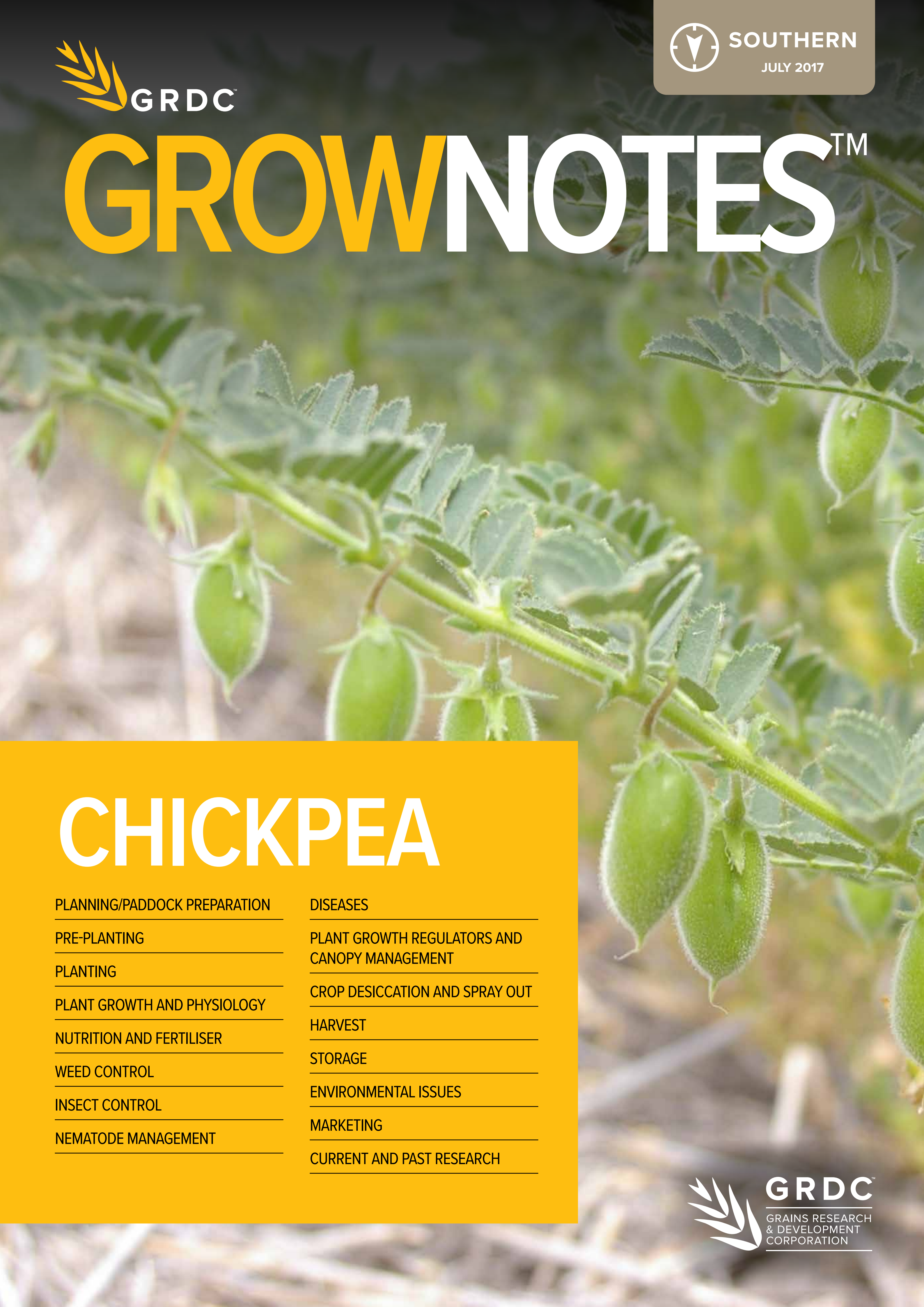

GRDC

SOUTHERN
JULY 2017

GROWNOTES™

CHICKPEA

GRDC
GRAINS RESEARCH & DEVELOPMENT CORPORATION

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Start here for answers to FAQ on chickpea crop management issues

What kind of chickpea best suits my area?

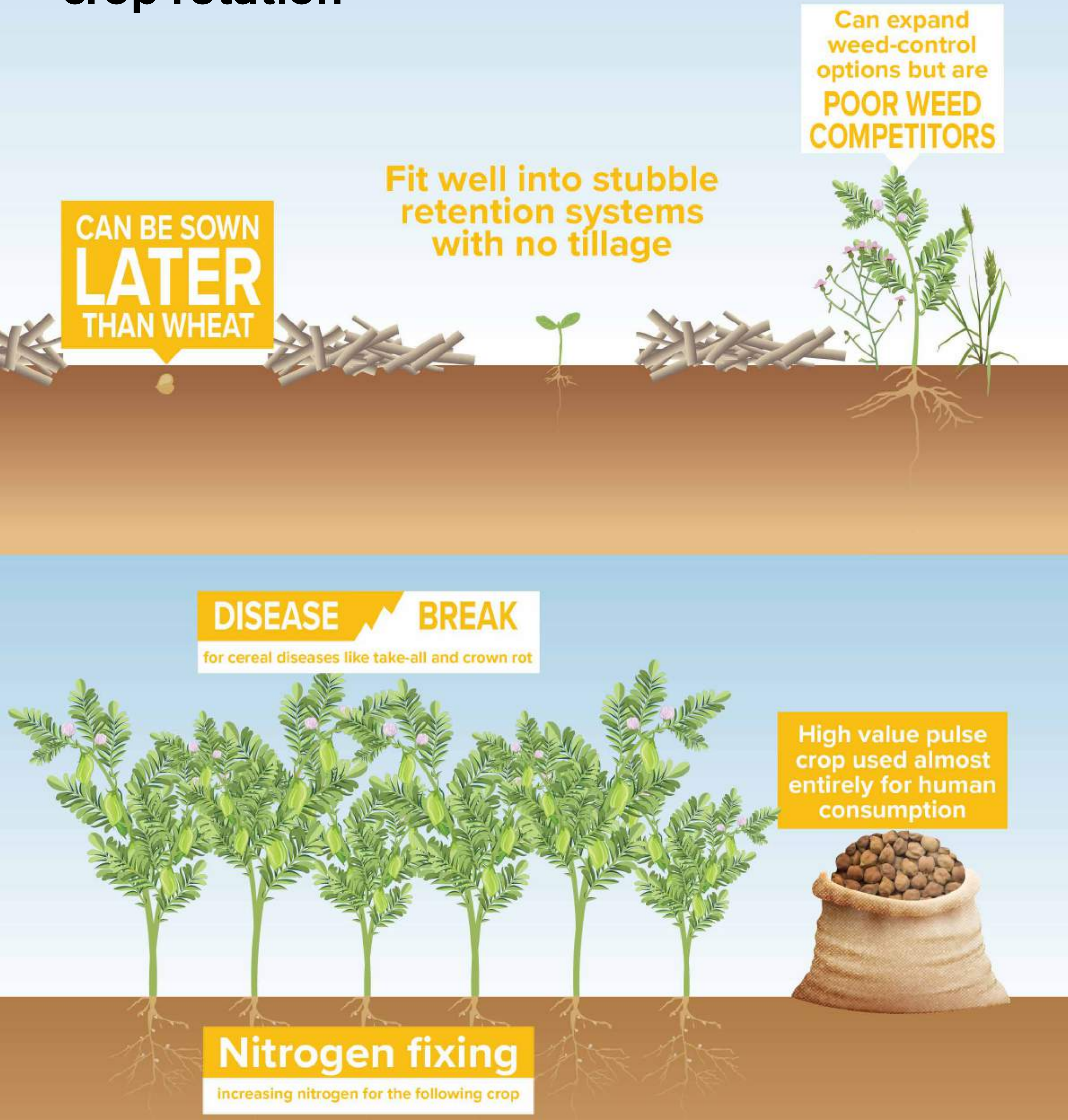
What can I do to minimise the impact of Ascochyta blight?

Do I need to apply any nitrogen to a chickpea crop?

What weed control options are there for chickpea crops?

Can I crop top or desiccate chickpea before harvest?

The benefits of including chickpeas in crop rotation



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Introduction

Key messages

- Chickpeas are an annual leguminous crop, used for human and animal consumption.
- There are two groups of chickpeas grown in Australia: Desi and Kabuli.
- Pulse crops, including chickpeas, tend to be grown in the medium to low rainfall (300-500 millimetres (mm)).
- Chickpeas prefer warmer growing conditions; average temperatures below 15°C will reduce pollen viability and can cause flower drop, and average temperatures over 35°C will lower the potential yield and cause possible flower abortion. Therefore, timing of sowing is very important for high yield harvests.
- Chickpeas are a very good source of carbohydrates and proteins, which together constitute about 80% of the total dry seed weight.
- New higher yielding varieties with improved resistance to Ascochyta blight have now been developed and should help to stimulate chickpea plantings. [1]

A.1 Crop overview

Chickpea (*Cicer arietinum*) is the second most important cool-season food legume worldwide, and is the largest pulse crop in Australia after lupin in terms of planting area and production (Figure 1). On average, chickpeas are sown on 41,1 000 ha annually to produce 448,000 t, with an average yield of 1.15 t/ha. [2] Chickpeas prices surged in early 2016. [3]

1 Agriculture Victoria. Growing Chickpea. http://agriculture.vic.gov.au/agriculture/grains-and-other-crops/crop-production/growing-chickpea

2 Reen, R. A., Thompson, J. P., Clewett, T. G., Sheedy, J. G., & Bell, K. L. (2014). Yield response in chickpea cultivars and wheat following crop rotations affecting population densities of Pratylenchus thornei and arbuscular mycorrhizal fungi. *Crop and Pasture Science*, *65*(5), 428-441.

3 A Felton-Taylor. ABC Rural. Chickpea price surges while disease risk is highlighted. http://www.abc.net.au/news/2016-04-06/chickpeas-set-to-surge-in-queensland/7303452

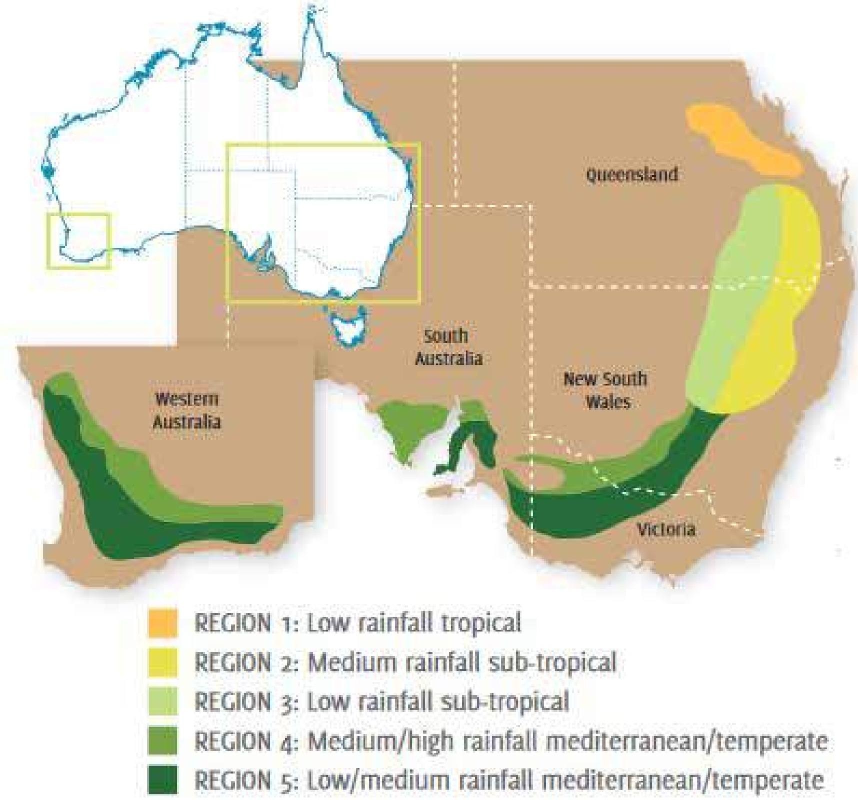


Figure 1: *Main growing areas for chickpeas in Australia. Pulse Breeding Australia (PBA) categorises chickpea production areas into five regions based on rainfall and geographic location.*

Source: Pulse Australia.

Chickpeas are an annual leguminous crop, with its grain used for human and animal consumption. There are two groups of chickpeas grown in Australia, Desi and Kabuli, mainly distinguished by seed size, shape, and colour. They also have different growth requirements, markets and end-users (Photo 1).

Photo 1: *Sales samples of pulses including split lentils, whole polished lentils, desi chickpeas and split field peas (being pointed to) at the Mumbai pulse markets, India.*

Source: GRDC.

Desi

Desi types have small angular seeds weighing about 120 mg, are wrinkled at the beak and range in colour from brown to light brown and fawn. They are normally dehulled and split to obtain dhal (Photo 2) and are favoured in the Asian sub-continent. Desi types are generally earlier maturing and higher yielding than the Kabuli types, particularly the larger seeded Kabulis. There is an increasing use of large, whole seeded desi types in a range of food preparations in Bangladesh. A small premium has been paid for Desi types (e.g. Kyabra) fitting this use. Desi chickpeas have traditionally made up about 90% to 95% of Australian production. [4]

Photo 2: *Desi Chickpeas (left) are split (right) to make dhal.*

Photos: DAFWA and MaGlobal.

Kabuli

Kabuli have larger, rounder seeds, weighing about 400 mg (Photo 3). They are white–cream in colour and are almost exclusively used whole. They are preferred through the Mediterranean region. They are sold whole, so seed size and appearance are critically important. Yields are generally lower and more variable than Desi varieties, although premiums for larger chickpeas can offset the yield disadvantage. Advances through plant breeding are giving more consistent results from Kabuli varieties. Kabuli seed sizes of 7–8 mm can command price premiums of >$100 per tonne (t) over Desi types, and sizes >8 mm considerably more. Most of Australia's Kabuli production occurs in the southern grain growing region. [5]

Photo 3: *Kabuli chickpeas produce larger seeds than desi.*

Source: Australian Agricultural Crop Technologies.

The plant is erect and freestanding, ranging in height from 40–60 cm, although well-grown plants may reach 80 cm. They have a fibrous taproot system, a number of woody stems forming from the base, upper secondary branches, and fine, frond-like

4 Pulse Australia. Chickpea Production: Southern and Western Regions. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

5 Pulse Australia. Chickpea Production: Southern and Western Regions. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

leaves. Each leaflet has a thick covering of glandular hairs that secrete a strong acid (malic), particularly during pod-set, and this provides some protection from insects. The plant can derive >70% of its nitrogen from symbiotic nitrogen fixation. Yields are best in areas with reliable seasonal rainfall and mild spring conditions during seed filling. Chickpeas are well suited to well-drained, non-acidic soils of a medium to heavy texture. [6]

Chickpeas are prepared and eaten in a variety of ways (Photo 4). Chickpeas are a staple food in the Middle East and the Indian subcontinent. The consumption of pulses in the western world is increasing as diets are becoming more diverse and people are recognising pulses' nutritional value. However, this is still a very small percentage of global consumption. Only 1% of Australian chickpeas is consumed locally, with the remaining percentage exported.

Photo 4: *Chickpeas are exported for human consumption.*

Source: Pulse.org.

A.1.1 Pulses

Chickpeas are pulses, which are annual legume crops that fix nitrogen from the atmosphere and produce high-protein grain for human consumption. [7] Pulses do not include green beans and peas; these are considered vegetable crops. Crops grown mainly for oil extraction (e.g. peanuts and soybean) are also excluded. Pulse crops tend to be grown in the medium to low rainfall (300-500 mm) environments with chickpea sown at the start of the growing season in May.

6 Pulse Australia. Chickpea, (*Cicer arietinum*). http://www.pulseaus.com.au/growing-pulses/bmp/chickpea

7 E Armstrong (2013) The role of pulses and their management in southern NSW. GRDC Update Papers 31 July 2013, http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/07/The-role-of-pulses-and-their-management-in-southern-NSW

VIDEOS

1. GCTV9: Southern Pulse.

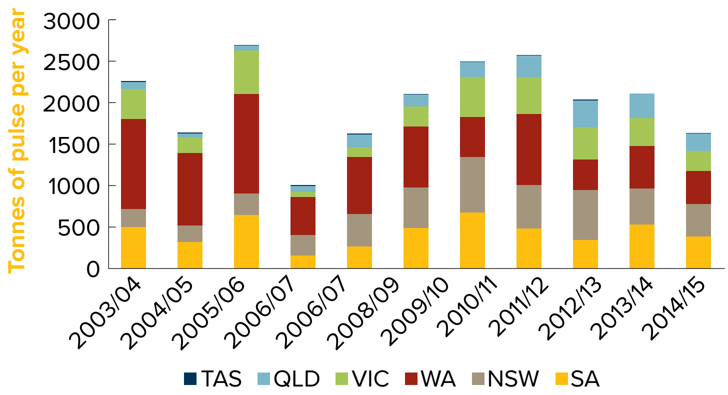


Figure 2: *Australian pulse production (tonnes of pulse per year).*

Source: ABARES and Pulse Australia.

Australian pulse production has increased rapidly over the last 30 years as a result of the appreciation by farmers of the financial returns from growing these crops and the role of pulses in sustainable cropping rotations. The area sown to pulses has increased from almost nothing in 1965 to over two million hectares in 2012, or about 10% of the area cropped (Figure 2). As part of crop rotation, chickpea has helped alleviate problems caused by continuous cereal cropping such as fungal root diseases, the depletion of soil organic matter and nitrogen levels, degradation of soil structure, and herbicide resistance in weeds. [8]

Pulse crops are generally sown in winter and harvested in late spring or summer. Chickpeas are grown in South Australia, Victoria, Western Australia, New South Wales, and Queensland. The majority of Australian-produced chickpeas are exported, with India, Pakistan and Bangladesh taking nearly 80% of all exported chickpeas. Chickpeas are suitable for both ruminant and non-ruminant feeds but are not commonly used for these purposes because of the higher prices obtained from human consumption markets. [9]

A.1.2 Quality attributes

Australian chickpeas are exported to more than 40 countries. The industry is committed to supplying chickpea with quality attributes tailored to these markets. Important quality traits targeted by chickpea breeders include:

- large and uniform seed size
- lighter coloured seed coat
- splitting quality of Desi chickpea
- hydration and cooking characteristics of Desi and Kabuli chickpeas[10]

A.1.3 Nutritional information

Chickpeas are a very good source of carbohydrates and proteins, which together constitute about 80% of the total dry seed weight (Table 1). Pulses are the major source of protein in vegetarian diets. Pulses have a protein percentage of 20–25%, compared with wheat, which has only half this amount and rice, only one-third this

8 Siddique, K. H. M., Brinsmead, R. B., Knight, R., Knights, E. J., Paull, J. G., & Rose, I. A. (2000). Adaptation of chickpea (*Cicer arietinum* L.) and faba bean (Vicia faba L.) to Australia. In *Linking research and marketing opportunities for pulses in the 21st century* (pp. 289-303). Springer Netherlands.

9 P Chudleigh (2012) An economic analysis of GRDC investment in the National Chickpea Breeding Program. GRDC Impact Assessment Report Series, December 2012, https://grdc.com.au/about/our-investment-process/impact-assessment

10 Pulse Australia (2010) A snapshot of Australian pulses. Poster reprint from CICILS/IPTIC Convention, http://www.pulseaus.com.au/storage/app/media/crops/2010_Australian-pulses.pdf

amount. [11] Starch, which is the principal carbohydrate component, varies in content from 41–50% and is lower in Desi varieties than in Kabuli varieties. Total seed carbohydrates vary from 52–71%. The crude protein content of chickpea varieties ranges from 16– 24%. Crude fibre, an important constituent of chickpeas, is mostly located within the seed coat. Based on amino acid composition, the proteins of chickpea seed were found, on average, to be of higher nutritive value than those of other grain legumes. Chickpeas meet adult human requirements for all essential amino acids except methionine and cysteine, and have a low level of tryptophan. Chickpeas have a high protein digestibility and are richer in phosphorus and calcium than other pulses.

Table 1: *Nutritional information for pulses per 100 g raw. These values should be taken as guidelines only; values can vary with variety, conditions of growth and age of pulse.*

	Chickpea	Field pea	Lupin	Lentil (red)	Lentil (green)	Faba bean	Mungbean
Energy (kJ)	986	886	1840	968	1550	1680	1800
Protein (g)	13	18	32	14	27	25	26
Fat (g)	3.8	0.8	5	0.4	2.5	1.3	2
Carbohydrate	41	40	26	44	58	57	72
Fibre (g)	17	19	15	7	10	8	12

Source: Pulse Australia.

A.2 Agronomy at a glance

- Avoid saline or sodic soils.
- Assess the Phytophthora risk.
- Avoid waterlogged areas.
- Control broadleaf weeds.
- Ensure there are no damaging levels of herbicide residue.
- Avoid planting near old chickpea stubble.
- Research variety choice and specific variety management packages.
- Ensure seed quality and seed fungicide dressing is adequate.
- Ensure inoculation procedures are adequate.
- Ensure fertiliser requirements are met.
- Assess crop establishment conditions.
- Monitor crops at critical stages.
- Respond to crop management needs in timely way.
- Set up boom spray for fungicides.
- Consider desiccation as harvest aide.
- Prepare storage infrastructure for grain at 14–16% moisture. [12]

A.3 Keys to successful chickpea production

- Make chickpeas part of an integrated cropping system involving wheat, canola, or barley. By taking a systems perspective and assessing financial performance over several seasons, you can estimate the true benefits of chickpeas in a rotation.
- Choose the right variety based on long term yield data for your region, maturity, disease resistance and the market opportunities for human consumption.

11 DAFF (2012) Chickpea—overview. Department of Agriculture Fisheries and Forestry Queensland, http://www.daff.qld.gov.au/plants/field-crops-and-pastures/broadacre-field-crops/chickpeas/overview

12 Pulse Australia, Checklist for Northern Growers http://www.pulseaus.com.au/storage/app/media/crops/2011_NPB-Chickpea-quality-seed.pdf

- Use good quality seed with high germination (80%+) and vigour that is free from seed-borne Ascochyta and Botrytis infection.
- Select and manage paddocks well in advance to control weeds and retain crop residues. Select paddocks with free draining soil of more neutral pH and low sodicity, salinity, and boron toxicity. Herbicide residues need to be considered, as well as likely weed pressure and the ability to effectively control weeds before seeding and in-crop.
- Establish sufficient plants as recommended for the variety, situation and sowing time. Ideally ground cover should coincide with the time pod filling begins. Plant populations can range from 30–50 plants/m^2 for rain-fed crops or higher if sown late.
- Sow on time. Sowing time is critical and regional recommendations should be followed. Late sowing reduces yield potential by lowering crop biomass, shortening the pod-filling period and increasing risks from moisture stress and high temperatures. Early sowing can lead to bulky crops, poor early pod set, and greater disease risks.
- Ensure good nutrition. Nitrogen (N) fertiliser is unnecessary as the crop's N requirement will be met through symbiotic fixing of atmospheric nitrogen, provided the seed is inoculated at sowing with the correct commercially available rhizobia inoculant. On phosphorus (P) deficient country, apply fertiliser at rates similar to or slightly less than those for wheat. On alkaline clay soils, fertilising with zinc may be warranted. On acid soils, molybdenum may be deficient and should be applied at or before sowing.
- Know the disease threats to chickpea and how to manage them for your district. No varieties are resistant to all fungal and virus diseases. The major risks are Ascochyta blight, Botrytis grey mould, and plant viruses. The impact of fungal diseases on yield can be diminished through the strategic selection and use of fungicides and crop management. Maintain an isolation distance of more than 500 m from the previous year's chickpea stubble, and eradicate volunteer plants over summer and autumn as these plants are a source of aphids (potential plant virus vectors) and disease inoculum.
- Control insect pests such as aphids and *Helicoverpa*. Controlling the 'green bridge' of weeds and volunteer crop plants over summer can reduce aphid populations and reduce the numbers that can infest the crop in the early growth stages. Monitoring for *Helicoverpa* (native budworm) caterpillars is essential during podding. Caterpillar feeding during podding will affect yield and potentially render the seed unsuitable for human consumption markets.
- Harvest on time, with a properly set-up header. Start harvesting when the seeds in the majority of pods 'rattle'. Stems may not be completely dry at this stage. Pods will thresh easily to yield clean, whole seeds with a minimum of splits and cracks provided the header settings are correct.
- Have a marketing plan that includes plans for on-farm storage or delivery options off-header. Investigate forward contracting if storage, pools, or warehousing is not an option.
- Optimise irrigation set-up and timing. Chickpeas respond well to irrigation in dry areas. Furrow irrigation is preferred over over-head irrigation. If necessary, pre-water then sow. To maximise yield potential, irrigate crops to produce maximum biomass while avoiding over-watering, as chickpea crops will not tolerate waterlogging. Do not allow the plants to stress during flowering and pod-fill. [13]

A.4 Brief history

The first grain legumes to be introduced into Australia were most likely field pea (*Pisum sativum L.*) and chickpeas (*Cicer arietinum L.*) in the late 19th century. Field pea has been grown ever since, albeit on a limited scale until its resurgence in the 1980s, but chickpeas remained ignored for almost 80 years. Market demand was low, with

13 Pulse Australia. Chickpea Production: Southern and Western Regions. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

limited consumption of grain legumes as food in Australia at that time and a restricted knowledge of trade opportunities. With a humble beginning of <0.08 million ha in 1971, winter grain legumes reached >2.3 million ha in the 2012 season (Figure 3). [14]

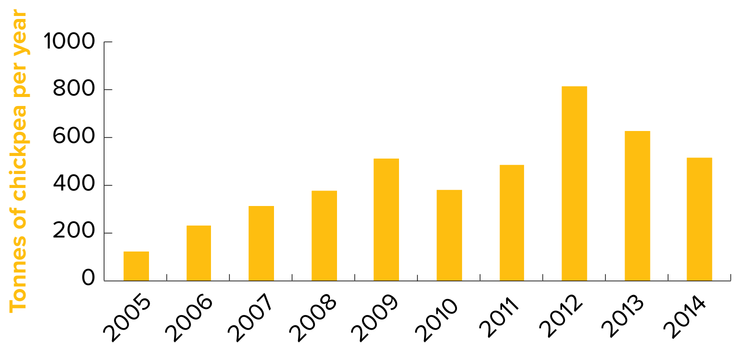


Figure 3: *Australia Chickpea production (Desi and Kabuli), tonnes of chickpea per year.*

Source: ABARES and Pulse Australia.

Chickpeas were first grown in Australia as a commercial crop in Goondiwindi, Queensland, during the early 1970s. The mixed cropping zone of dryland agriculture of south-eastern Australia traditionally raised cereal crops for 2–5 years followed by a matching or greater period of grazed, legume-based pastures. Chickpeas were first grown in the southern region during the 1980s with the sown area peaking in the mid-1990s before Ascochyta blight almost wiped the industry out. New varieties of chickpeas that are Ascochyta-resistant are now available and the industry is growing once again. [15]

The amount of chickpeas grown in the southern growing region has varied over the last five years (Figures 4-7).

14 Siddique, K. H. M., Erskine, W., Hobson, K., Knights, E. J., Leonforte, A., Khan, T. N., ... & Materne, M. (2013). Cool-season grain legume improvement in Australia—use of genetic resources. *Crop and Pasture Science*, *64*(4), 347-360.

15 Pulse Australia. Chickpea Production: Southern and Western Region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

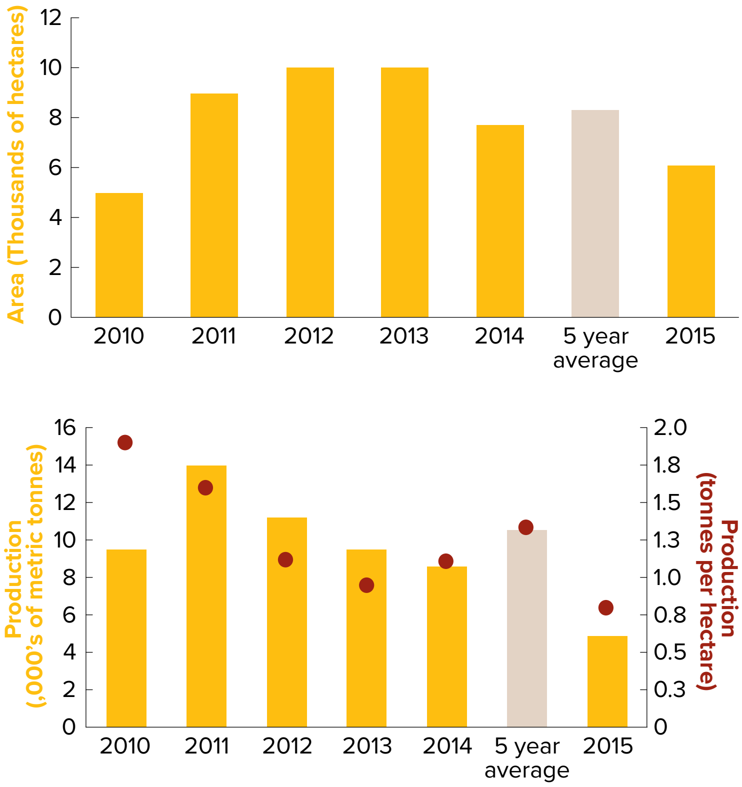


Figure 4: *Area sown to Desi chickpeas (top) and production in metric tonnes (bottom) in Victoria.*

Source: Pulse Australia.

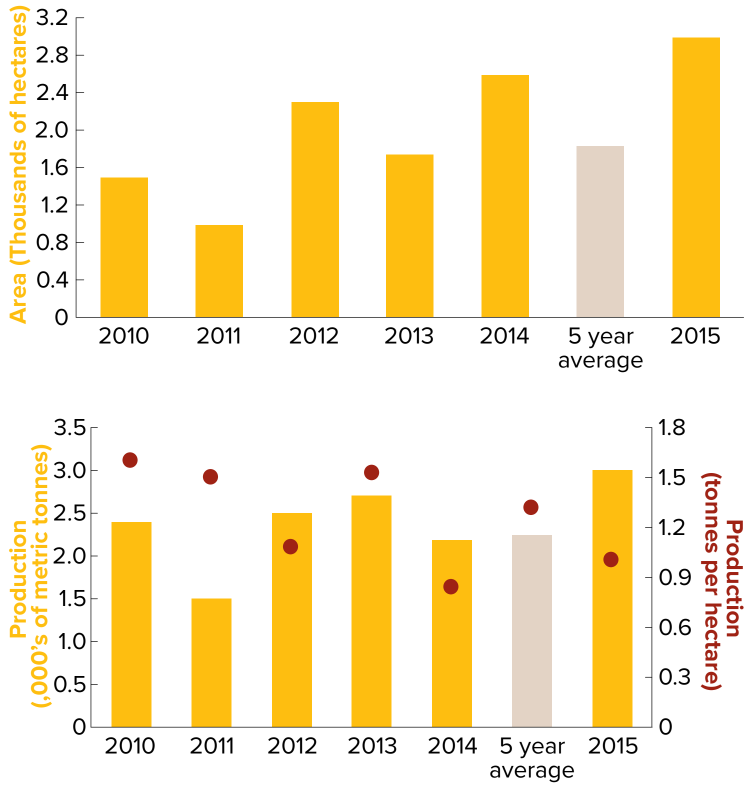


Figure 5: *Area sown to Desi chickpeas (top) and production in metric tonnes (bottom) in South Australia.*

Source: Pulse Australia.

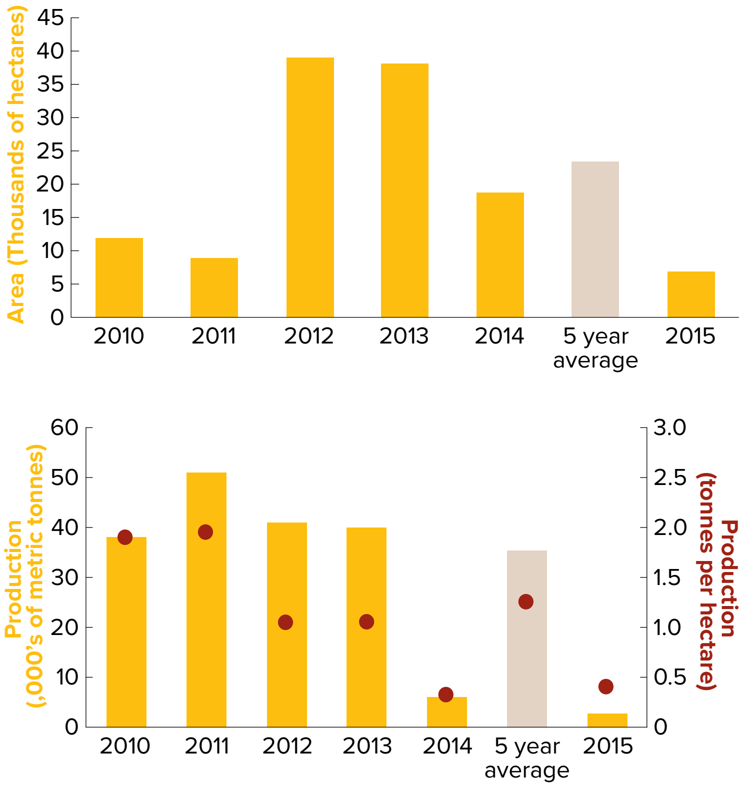
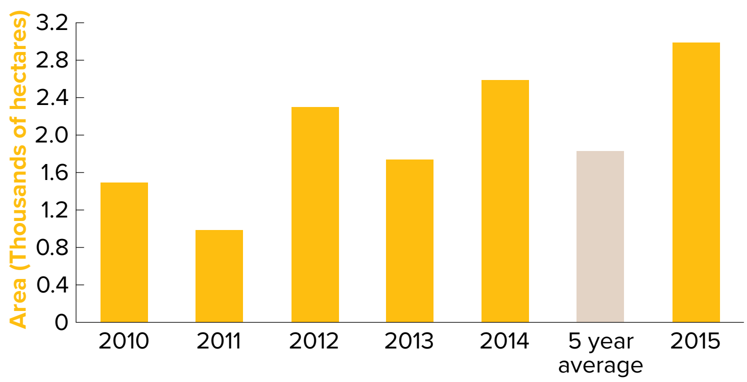


Figure 6: *Area sown to Kabuli chickpeas (top) and production in metric tonnes (bottom) in Victoria.*

Source: Pulse Australia.



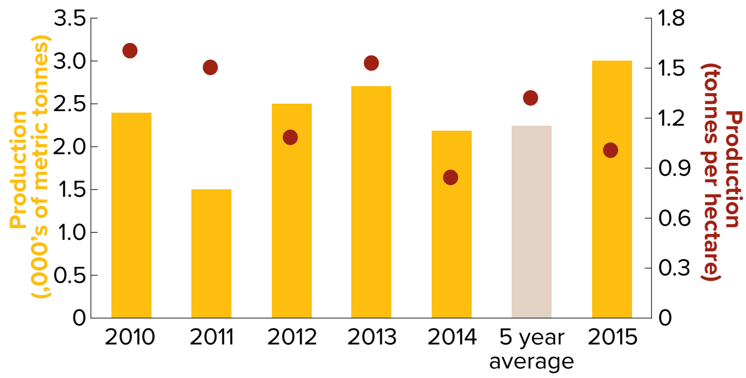


Figure 7: *Area sown to Kabuli chickpeas (top) and production in metric tonnes (bottom) in South Australia.*

Source: Pulse Australia.

Table 2: *Chickpea crop production Ha and Tonne for 2015-2016 in South Australia.*

	Lower Eyre Peninsula	Eastern Eyre Peninsula	Yorke Peninsula	Upper North	Mid North	Lower North	Central hills and Fleurieu	Lower Murray	Nth Murray Mallee	Sth Murray Mallee	Upper south east	Lower south east	State total
Ha	400	200	6500	3200	5000	1000	200	200	1400	1000	200	200	19500
t	500	100	8500	3500	5500	1300	250	200	1000	900	300	250	22300

Source: PIR.SA.

2016 saw South Australia's first line of cooked, ready-to-serve chickpeas, as an alternative to imported canned products. The chickpeas are sourced from a collection of growers from the mid-north of South Australia to the York Peninsula (Photo 5). [16]

16 In Daily, Adelaide. Ready-to-eat chickpeas a first for SA. http://indaily.com.au/eat-drink-explore/2016/02/09/ready-to-eat-chickpeas-a-first-for-sa/

Photo 5: *The first line of chickpeas grown and packaged in Southern Australia.*

Source: In Daily.

A.5 GRDC Chickpea breeding investment

The chickpea breeding program has so far relied on conventional breeding techniques. However, the amount of genetic and genomic resources is increasing, and in the near future, marker-assisted breeding methods will be deployed for traits such as pyramiding minor genes for Ascochyta blight resistance and combining these with high levels of PRR resistance. It is anticipated that the use of such technologies will be more economical and allow quicker delivery of varieties to Australian growers than current methodologies. [17]

The principal outputs of GRDC chickpea-breeding investments have been improved varieties (Photo 6). Important traits from these improved varieties have been disease and pest resistance and traits that influence yield. Improvements in these traits were delivered in the new varieties released between 2005 and 2012. Higher yields and increased disease resistance can translate into higher profits from the chickpea crop, in turn potentially increasing the attractiveness of chickpeas in a cereal rotation and benefiting the next cereal crop.

17 Siddique, K. H. M., Erskine, W., Hobson, K., Knights, E. J., Leonforte, A., Khan, T. N., ... & Materne, M. (2013). Cool-season grain legume improvement in Australia—use of genetic resources. *Crop and Pasture Science*, *64*(4), 347-360.

Photo 6: *The GRDC-funded chickpea-breeding program has resulted in improved varieties with better disease and pest resistance.*

Source: NSW DPI.

GRDC's investment in three projects (including the Australian Chickpea breeding program (DAN00094), PBA Chickpea – National breeding program (DAN00151) and DAN00065) is expected to produce a number of benefits. The total investment of $43 million has been estimated to produce total gross benefits of $123 million, providing a net present value of $80 million, a benefit–cost ratio of just under 3 to 1 (over 30 years, using a 5% discount rate), and an internal rate of return of >15%. [18]

Pulse Breeding Australian (PBA) is a world-class Australian breeding program for chickpeas, field peas, faba beans, lentils, and lupins. PBA has operated since 2006 and its vision is to see pulses expand to >15% of the cropping area so as to underpin the productivity, profitability, and sustainability of Australian grain farming systems. PBA is developing a pipeline of improved varieties for Australian growers that achieve higher yields, have resistance to major diseases and stresses, and have grain qualities that enhance market competitiveness.

PBA is an unincorporated joint venture between:

Department of Primary Industries, Victoria (DPI Vic)

South Australian Research and Development Institute (SARDI)

Department of Agriculture, Fisheries and Forestry, Queensland (DAFF Qld)

New South Wales Department of Primary Industries (NSW DPI)

Department of Agriculture and Food Western Australia (DAFWA)

University of Adelaide

University of Sydney

Pulse Australia

Grains Research and Development Corporation (GRDC)

A.6 Keywords

Chickpeas, Desi, Kabuli, pulse, nitrogen fixation, rotation, breeding.

VIDEOS

2. GCTV4: Pulse Breeding Australia Retrospective

18 P Chudleigh (2012) An economic analysis of GRDC investment in the National Chickpea Breeding Program. GRDC Impact Assessment Report Series, December 2012, https://grdc.com.au/about/our-investment-process/impact-assessment

Planning/Paddock preparation

Key messages

- To reduce disease risk, chickpea crops should be separated from the previous year's crop by at least 500 m and up to 1 km in areas where old stubble is prone to movement, i.e. downslope and on flood plains.
- Avoid paddocks with high weed burdens, as chickpeas provide poor competition for weeds.
- Chickpea is well suited as a winter crop to the medium rainfall (300–500 mm) areas of south-eastern Australia. Crop growth during winter months is very limited but accelerates with warmer weather in spring (Figure 1).
- Kabuli types are less tolerant than Desi types to dry conditions, as they require more moisture to achieve a satisfactory grain size and yield. Desi types require above 350 mm annual rainfall and Kabuli types need more than 400 mm, though it's important to check with agronomist for local recommendations.
- Chickpea is an advantageous crop rotation because of its nitrogen fixing ability. However, it is a poor competitor against weeds and can be susceptible to nematodes.
- Consider herbicide residual and plant-back effects before sowing.
- Prior to sowing, it is important to consider disease, nematode, and pest management by knowing paddock history and testing soils.

Figure 1: *Ideal conditions for chickpea growth.*

Source: Pulse Australia.

1.1 Paddock selection

Key points

- Well-drained soils with a pH above 5.5 in Calcium Chloride ($CaCl_2$), heavy deep clays, heavy loam, sandy loam and Salmon Gum soils. Avoid soils with a pH below 5.5 ($CaCl_2$), saline soils, high boron soils and Wodjil soils. Chickpeas have poor tolerance of low pH, where aluminium toxicity can be a problem.
- Check soil tests to determine if the soil type is suitable for chickpea production, i.e. pH 5.2–8.0, loams to self-mulching clays, sufficient stored soil moisture, absence of herbicide residues and absence of constraints such as sodicity, salinity/chloride, high bulk density, and potential for waterlogging.
- Chickpeas grow poorly on sands, on tight hard-setting clays and acidic soils.
- A soil structure and slope which allows good drainage—avoid shallow soils.

MORE INFORMATION

Checklist for Chickpea growers in the Southern region.

VIDEOS

1. Growing profitable pulses.

- Check herbicide use over the previous 12–24 months and seek advice regarding any potential residue problems prior to sowing. Make sure there is little or no risk of sulfonylurea carry over.
- A low broad-leaf weed burden.
- Soil surface needs to be relatively flat and even after sowing for harvest.
- Chickpea crops should be separated from previous year's crop by at least 500 m and up to 1 km in areas where old stubble is prone to movement, i.e. down slope and on flood plains. This helps to reduce the spread of Ascochyta blight, a foliar/stubble-borne disease.
- A paddock with a history of medics or lucerne should also be avoided due to potential risk of phytophthora infection. Also consider past history of sclerotinia and rhizoctonia infection.

Aim to direct drill chickpeas into standing cereal stubble. Crops reliably yield 10% higher when established this way. Uniformity of soil type, paddock topography, and surface condition of the paddock are all important criteria in assessing whether country is suitable for chickpea production. Harvest losses are much higher in rough or uneven paddocks, particularly in dry seasons when crop height is reduced. Sticks or rocks, eroded gullies or gilgais ('melon' or 'crab' holes) will prevent headers operating at low cutting height. This is particularly important when using headers with wide fronts. Small variations in paddock topography can lead to big variations in cutting height across a wide front and a subsequent increase in harvest losses.

Moisture supply can significantly affect crop maturity. Changes in soil type and moisture holding capacity across a paddock can lead to uneven crop maturation, delayed harvesting, and increased risk of weather damage or high harvest losses. Paddocks that have even soil types are relatively easier to manage, and are preferred for chickpeas. [1]

Selecting a paddock with minimal variation in soil type will often help to provide even maturity and ripening of the crop. This will enable harvesting at the earliest possible time, increase quality, and minimise harvest losses. The overall result is usually a more profitable crop.

1.1.1 Avoid deep gilgai or heavily contoured country

Contours and undulating country ('melon holes' or 'crab holes') present two problems:

- Uneven crop maturity due to variation in soil water supply. Melon-holes usually store more water than the mounds, and the crop in wetter areas will often continue to flower and pod when the rest of the crop is already drying down. Similarly, contour banks retain more moisture after rain, and prolong crop maturity relative to the rest of the crop late in the crop.
- High harvest losses and increased risk of dirt contamination in the header sample. Many dryland chickpea crops require the header front to be set close to ground level, and even small variations in paddock topography can lead to large variations in cutting height across the header front, and a significant increase in harvest losses.

Contamination of the harvested sample with dirt and clods is difficult to avoid in undulating, gilgai country, and can cause a significant increase in grading losses and costs.

Foreign material must not exceed 3% by weight, of which no more than 0.3% must be unmillable material (soil, stones, and non-vegetable matter). If a farmer delivers chickpeas that do not meet this export standard, they will need to be graded at a cost of $15–25/t.

1 Pulse Australia. Chickpea production: Southern and Western Region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

1.1.2 Soil

Chickpea crops are best suited to well-drained loam and clay loam soils that are neutral to alkaline (pH 6.0–9.0) and have good water holding capacity.

Prior to planting, review soil tests and records, paying particular attention to the following soil characteristics:

- soil type—loams to self-mulching clays
- pH 5.2–8.0 - saline soils ECe >1.5 ds/m will cause a yield reduction
- salinity, chloride—avoid electrical conductivity (EC) levels >1.5 dS/m
- sodicity (ESP) >1.0 surface or >5.0 in subsoil can limit yield
- High boron and soil chloride levels >600 mg/kg in sub-soil layers will severely limit root growth, depth, and water extraction from the soil.
- potential waterlogging problems: Avoid dense soils (bulk density >1.5) or compacted soils or areas where free water does not drain away and/or remains saturated. Spring sowing may be an option in higher rainfall areas. [2]
- amount of stored soil moisture and received rainfall, noting their potential impact on herbicide residues[3]

For more information on causes and management of waterlogging, salinity, sodicity, and chloride issues, see Section 14: Environmental issues.

Chickpeas will not grow in light acid soils and areas prone to waterlogging should be avoided. Chickpea crops must be harvested close to the ground so stony paddocks or fields with uneven soil surface (e.g. gilgai) should be avoided.

Chickpea are susceptible to hostile sub-soils, with boron toxicity, sodicity, and salinity causing patchiness in affected paddocks. Chickpea will not tolerate soils with any exchangeable aluminium present. Tolerance to sodicity in the root zone (to 90 cm) is less than 1% exchangeable sodium (ESP) on the surface and less than 5% ESP in the subsoil. [4]

Subsoil

Chickpeas are recommended for soils with a surface pH ca of 5.0, if sub-soil pH rises to above 5.5 within 10–15 cm of the surface. [5]

Subsurface pH is of importance to the break crop in the rotation. Knowing the pH at depth is vital for an effective break crop, particularly if the surface soil is testing at or above the target of pH 5.5.

Crop yield variability and productivity below potential yield on neutral and alkaline soils in the semiarid Mediterranean-type environments of south-eastern Australia have been attributed, in part, to variable rooting depth and incomplete soil water extraction caused by physical and chemical characteristics of soil horizons below the surface. These characteristics are referred to as subsoil constraints. Information concerning subsoil constraints typical of neutral and alkaline soils in south-eastern Australia, principally salinity, sodicity, dense soils with high penetration resistance, waterlogging, nutrient deficiencies, and ion toxicities was recently reviewed. The review focused on information from Australia (published and unpublished), using overseas data only where no suitable Australian data is available.

An assessment of the effectiveness of current management options to address subsoil constraints is provided. These options are broadly grouped into three categories:

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2 Pulse Australia. Southern Pulse Bulletin PA 2010 #05 – Chickpea checklist for southern growers. http://www.pulseaus.com.au/storage/app/media/crops/2010_SPB-Chickpea-checklist-south.pdf

3 G Cumming (2014) Chickpea varieties selecting horses for courses. GRDC Update Papers 5 March 2014, http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Chickpea-varieties-selecting-horses-for-courses

4 Pulse Australia. Chickpea production: Southern and Western Region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

5 Parker W. DAFWA (2014). Crop Updates – Break crops being sown onto unsuitable soils, unsuspectingly.

(i) amelioration strategies, such as deep ripping, gypsum application or the use of polyacrylamides to reduce sodicity and/or bulk density, deep placement of nutrients or organic matter to overcome subsoil nutrient deficiencies, or the growing of 'primer' crops to naturally ameliorate the soil;

(ii) breeding initiatives for increased crop tolerance to toxicities such as salt and boron; and

(iii) avoidance through appropriate agronomic or agro-engineering solutions.

MORE INFORMATION

Subsoil constraints to crop production on neutral and alkaline soils in south-eastern Australia: a review of current knowledge and management strategies.

There a number of difficulties in identifying the impact of any single subsoil constraint to crop production on neutral and alkaline soils in south-eastern Australia, given that multiple constraints may be present. Difficulty in clearly ranking the relative effect of particular subsoil constraints on crop production (either between constraints or in relation to other edaphic and biological factors) limits current ability to develop targeted solutions designed to overcome these constraints. Furthermore, it is recognised that the task is complicated by spatial and temporal variability of soil physicochemical properties and nutrient availability, as well as other factors such as disease and drought stress. Nevertheless, knowledge of the relative importance of particular subsoil constraints to crop production, and an assessment of impact on crop productivity, are deemed critical to the development of potential management solutions for these neutral to alkaline soils. [6]

1.1.3 Stubble retention

Chickpeas fit well into stubble retention systems with no tillage, and serve their wider role in crop rotations and farming systems. Retention of adequate plant residues on the surface is important to protect the soil from erosion both during growth and after harvest. This will not affect chickpea germination and growth, and can improve establishment on hard-setting, surface-crusting soils. Sowing into cereal stubble reduces soil moisture losses from evaporation. [7]

Chickpeas established by direct drilling into standing cereal stubble reliably yield 10% higher than when using other planting techniques (Photo 1). [8]

6 Adcock, D., McNeill, A. M., McDonald, G. K., & Armstrong, R. D. (2007). Subsoil constraints to crop production on neutral and alkaline soils in south-eastern Australia: a review of current knowledge and management strategies. *Animal Production Science*, *47*(11), 1245-1261.

7 Pulses Australia. Chickpea Production: Southern and Western region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

8 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

VIDEOS

2. Stubble Management.

3. GCTV15: Stubble height Pt. 1.

4. GCTV15: Stubble height Pt. 2.

5. GCTV4: Burning for stubble retention.

6. Southern farm groups cutting through stubble issues.

GRDC Southern 'stubble initiative' workshop

Photo 1: *Better chickpea yields are achieved by sowing into cereal stubble.*

Source: Flickr.

There are advantages and disadvantage to stubble retention, with high stubble loads potentially causing problems for growers in the following year (Table 1).

Table 1: *Advantages and disadvantages in retaining stubble. Based on information from the GRDC.*

Advantages of stubble retention	Potential disadvantages of stubble retention
Retained stubble provides excellent ground cover (>2.5 t/ha), thus reducing wind and water erosion;	Seeder blockages impact on plant establishment;
increases rainfall infiltration;	Stubble provides ideal habitat for pests to survive;
reduces moisture evaporation (>4.5 t/ha);	physically intercepts herbicides;
eliminates the need to burn, bale or incorporate; and	increases frost risk;
recycles nutrients back into the soil.	carries over diseases; and
	increases potential for nitrogen tie-up.

For more detailed information, see Wide rows and stubble retention and Profitable stubble retention systems for the high rainfall zone.

Stubble and its impact on temperature in chickpea crops

Key points:

- Chickpea sown into flattened residue had lower (av. 1.0°C) minimum temperatures compared to standing residue.
- Chickpea sown into flattened residue had higher (av. 3.4°C) maximum temperatures compared to standing residue.
- Stubble thresholds are unknown at this stage.

Stubble affects soil physical properties such as temperature and moisture. The effect on temperature is due to landscape features such as whether a paddock was on top of a hill, on a hill slope, or at the lower end of a slope because cold air (due to its higher density) tends to flow downhill and settle in the lower parts of the landscape, leading to colder pockets where temperatures decline the most.

Stubble cover also affects air and soil temperature. During the day the stubble reflects radiation due to its 'albedo'. A bare, darker soil absorbs more solar radiation than a stubble-covered soil and warms up more readily. The stubble also acts as insulation—it contains a lot of air which is a poor conductor of heat.

Finally, the stubble affects the moisture content of the soil. It takes more heat to warm up moist, stubble-covered soil than dry, bare soil. This causes soil temperature of a bare soil to be higher than stubble-covered soil during the day (especially in the afternoon). At night, however, the bare soil loses more heat than stubble-covered soil due to the lack of insulation (the air-filled mulch being a poorer heat conductor). This is especially noticeable when skies are clear. The air above the bare soil is therefore warmer during the night than the stubble-covered surface. This can affect canopy temperature profiles in crops.

In a recent trial, the temperature of stubble and the subsequent impacts on chickpea crops in NSW were explored.

Temperature sensors were placed between 1.0 m wide rows in:

1. plots sown into standing stubble with bare soil between chickpea rows
2. plots sown into flattened stubble with surface stubble between chickpea rows

Standing stubble plots with bare soil between rows:

- had minimum temperatures 1°C warmer at the base of the canopy than surface-stubble plots during vegetative period
- had maximum temperatures -3.4°C cooler at the base of the canopy than surface-stubble plots during flowering and grain fill period
- recorded five days with maximum temperatures >35°C compared to 27 days of maximum temperatures >35°C where stubble was flattened.

Plant components for the stubble treatments are shown in Table 2. Plants sown into bare soil between standing wheat rows had higher grain yields which were achieved through more pods being set and more seeds being produced per square metre.

Table 2: *Effect of stubble treatment on selected plant components.*

Stubble	DM/m²(g)	Grain/m² (g)	Seeds/m²	Pod No/ m²	Seeds/ pod	HI
Bare soil	706	270	1072	815	1.3	0.38
Straw	526	226	908	538	1.7	0.43

Source: GRDC.

Conclusions

- Flattened surface residue led to lower minimum temperatures in crop than standing residue.
- Flattened residue had higher maximum temperatures during flowering and grain fill than standing residue.
- Flattening and spreading residue can increase crown rot infection in the following wheat crop.
- Keep wheat stubble standing in defined rows and sow chickpeas between wheat rows. [9]

1.1.4 Rainfall

Chickpeas are not well-suited to the lower rainfall, hotter areas, although the plants will set seed under warmer conditions where other pulse crops are likely to fail. Cool, wet conditions are more likely to stimulate foliar diseases and these can adversely affect seed set and yield. Desi varieties should only be grown in areas where the

VIDEOS

7. Crop sequencing in the Mallee.

9 Verrell, A. (2016). GRDC Update Papers - Stubble and its impact on temperature in chickpea crops. https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/07/Stubble-and-its-impact-on-temperature-in-chickpea-crops

annual rainfall is greater than 350 mm. Sowing is best carried out from early May to early June, with early sowing recommended for the lower rainfall areas. Kabuli varieties are later maturing and should only be grown in areas where the annual rainfall is over 400 mm. As a higher value crop, Kabuli types are often grown on fallow where extra moisture can mean larger seed. [10]

1.1.5 Physical constraints

Physical constraints decrease oxygen and water movement in soils. Compacted soils and those with high physical strength (bulk density >1.5 g/cm3) impede root growth. Dense, impermeable subsoils (high bulk density) can lead to extensive development of lateral roots in the top 30 cm of soil, with only weak development of the taproot. Subsoil compaction can be caused by heavy traffic or tillage on wet soils. Compacted layers may be visible, measured by high penetration resistance (>2 MPa), or indicated by distorted root growth. Deep ripping of soils and use of controlled traffic can help to overcome compaction, but in some soils, amelioration with organic matter, gypsum or lime, for example, may be required as well. Chickpeas are particularly prone to hard pans and compacted soils, and suffer more from waterlogging if compaction layers exist.

1.1.6 Nutrient constraints

Crop management can affect nutrient deficiencies. Iron deficiency in pulses is more likely to occur in wheel tracks and compacted areas. Manganese deficiency is more likely in light, fluffy soil.

In pulses, cobalt and molybdenum are required for nodulation and nitrogen fixation, so deficiency of these trace elements can lead to poor nodulation.

For more information, see Section 5: Nutrition and Fertiliser.

1.1.7 Biological constraints

Problems can occur when there is a lack of beneficial organisms such as earthworms and arbuscular mycorrhizae fungi (AMF) in soils. Their build-up can be encouraged by use of stubble retention and direct drilling or no tillage as well as appropriate crop rotations.

Biological inputs

Key points:

- When evaluating a biological input for grain production, it may be useful to consider whether the input will alleviate yield constraints.
- The major yield constraints in the southern grain-growing region are high soil density, sodicity, and acidity.
- The biological inputs with the most potential to help alleviate these yield constraints are manure, compost, vermicompost, biochar, and some biostimulants.

The southern region has soils with generally low fertility and many have subsoil constraints, such as salinity, sodicity and toxic levels of some elements. However, soils in this region are diverse and some areas have very productive soils. Crop production systems in the region are varied and include many mixed farming enterprises that have significant livestock and cropping activities.

Yield potential in the southern region depends on seasonal rainfall, especially in autumn and spring, and there is less dependence on stored soil moisture than in the northern region.

In the southern grain-growing region, the most significant yield constraints are high soil density, sodicity, and acidity.

10 Pulse Australia. Chickpea production: Southern and Western Region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

Alleviating yield constraints using biological inputs

When the high density of a soil constrains yield, applying manure, compost, vermicompost or biochar may increase crop growth and yield. Because these biological inputs are largely composed of organic matter, they can increase soil aggregation and pore space in soil, both of which can decrease soil compaction and the bulk density of soil. However, if yield is being constrained by subsoil compaction, manure, compost or biochar are unlikely to alleviate this constraint unless they are applied below the soil surface or incorporated into soil.

When yield is constrained by soil sodicity, crop growth and yield may be increased by applying manure, compost, vermicompost or biochar. These biological inputs do not actually decrease the sodicity of soil. Instead, the high organic matter content of these inputs means that they may help alleviate the poor structural properties associated with sodic soils. However, when sodicity constrains yield, the simplest and most economic way of alleviating this constraint is applying gypsum to soil.

When soil acidity constrains yield, applying compost or biochar may increase crop growth and yield. Compost and biochar both have the potential to increase the pH of acidic soils. Compost can also have a strong pH buffering capacity, which can help minimise future changes in soil pH. Although compost and biochar can increase soil pH, when soil acidity constrains yield, the simplest and most economic way of alleviating this constraint is applying lime to soil. [11]

1.1.8 Problematic paddocks

Stones and sticks are a concern in poorly or recently cleared country if not able to be rolled in. Harvest losses increase dramatically if the header front needs to be raised to avoid serious mechanical damage to the header. Small stones and wood fragments can also contaminate the seed sample and downgrade quality.

Cloddy or badly ridged paddocks are likely to cause contamination of the chickpea sample during harvest. Level the soil surface as much as possible, either during ground preparation or at sowing. A land-roller can be helpful after sowing, in cultivated situations, to level the soil surface and push clods of soil and small stones back down to level with the surface.

1.1.9 Soil pH

Key points:

- Soil pH is a measure of the concentration of hydrogen ions in the soil solution. It is measured in water or $CACl^2$ solution, providing slightly different readings.
- Low pH values (<5.5) indicate acidic soils and high pH values (>8.0) indicate alkaline soils.
- Soil pH between 5.5 and 8 is not usually a constraint to crop or pasture production.
- In South Australia, more than 60% of agricultural soils are alkaline.
- Outside of the optimal soil pH range, microelement toxicity damages crops.

Hydrogen ion concentration in the soil is called pH and is influenced by chemical reactions between soil components and water. Soil pH is affected by the varied combinations of positively charged ions (sodium, potassium, magnesium, calcium, aluminum, manganese, and iron) and negatively charged ions (sulfate, chloride, bicarbonate, and carbonate). Soil pH directly affects the concentration of major nutrients and the forms of microelements available for plant uptake and can result in deficiencies or toxicities (Figure 2).

11 Soilquality.org. Biological inputs – Southern Grain growing region. http://www.soilquality.org.au/factsheets/biological-inputs-southern-grain-growing-region

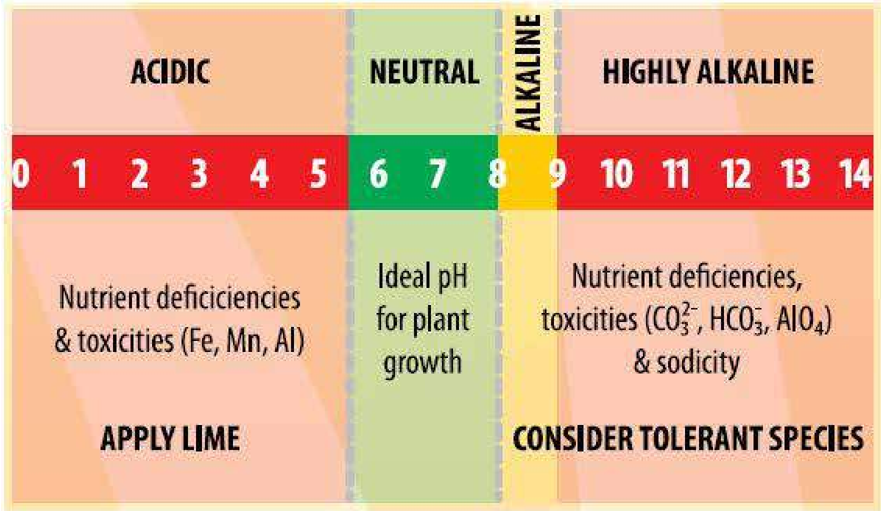


Figure 2: *Classification of soils on the basis of pH (1:5 soil:water), the implications for plant growth and some management options.*

Source: Soilquality.org.

Alkaline soils

Soil alkalinity is mainly caused by bicarbonates and carbonates, although phosphates, borates, and some organic molecules can contribute. In a soil with pH from 7–8.2, bicarbonates and carbonates of calcium and magnesium dominate. Calcareous soils contain from 1–90% lime material as calcium carbonates and these sparingly soluble salts cause the soil to have a pH of 8.0–8.2, which is not a severe problem for plant growth or agricultural production. Problems are encountered in alkaline soils when sodium occurs or accumulates and forms salts such as sodium bicarbonate and sodium carbonate. These are highly soluble and increase the soil pH above 8. When the pH is more than 9, the soils are considered highly alkaline and often have toxic amounts of bicarbonate, carbonate, aluminium, and iron. Nutrient deficiency is also likely to be a major problem and the high amount of exchangeable sodium in these soils reduces soil fertility.

Managing soil pH

Acid soils

Acid soils can be economically managed by the addition of agricultural lime, usually crushed limestone. Sufficient lime should be added to raise the pH to above 5.5. The amount of lime required to ameliorate acid soils will vary, mainly depending on the quality of the lime, soil type, and how acidic the soil has become.

Soils prone to becoming acidic will need liming every few years. Seek advice on an appropriate liming regime from your local agricultural advisor.

Alkaline soils

Treating alkaline soils by the addition of acidifying agents is not generally a feasible option due to the large buffering capacity of soils and uneconomic amounts of acidifying agent (e.g. sulfuric acid, elemental sulfur, or pyrites) required.

Gypsum will reduce sodicity and this can reduce alkaline pH to some extent. Growing legumes in crop rotation may help in sustaining any pH reduction.

In high pH soils, using alkalinity tolerant varieties of crops and pasture can reduce the impact of high pH. [12]

12 Soiquality.org. Soil pH- South Australia. http://www.soilquality.org.au/factsheets/soil-ph-south-austral

1.1.10 Bunching and clumping of stubble

Stubble bunching or clumping can occur when sowing into retained stubble as a result of blockages during sowing. These mounds of stubble are often picked up in the header front, causing mechanical blockages and contamination of the sample if they contain excessive amounts of soil.

Management options for dealing with stubble clumping include:

- use of a no-till (disc) seeder or other seeder capable of handling heavy stubble
- modification of existing air-seeders (tine shape and lifting some tines)
- sowing before soil and stubble becomes too wet
- use of rotary harrows to spread and level stubble and sow between old plant rows aiming to leave stubble standing.

Standing stubble can be slashed or burnt if sowing equipment with good trash flow is not available. [13]

1.1.11 Soil testing

Key points:

- The approach taken will be defined by the purpose of the investigation, variability in the area sampled, and the analysis and accuracy required.
- For many soil quality parameters, sampling is typically done to 10 cm. Although 30 cm is required for carbon accounting purposes, stratification below 10 cm is recommended (e.g. 10–20, 20–30 cm).
- The sampling strategy should either integrate or describe the variation within the sampling area.
- Samples should be air dried or kept below 4°C prior to analysis. For biological measurements, it is best to analyse as soon as possible.

Before you can decide how you are going to soil sample, you need to be clear about the purpose of your sampling. Different sampling approaches may be required depending on what you are sampling for, the soil type, the management unit (e.g. paddock), soil spatial variability (changes in soil type, dunes-swales etc.), the accuracy required of the result, and the value given to the information provided (Photo 2). So before you start, define very clearly the question you are asking of your soil samples.

Consult a professional soil scientist, agronomist, or your analytical laboratory to be sure that your soil samples are taken at the right time, from the right depth, the right place, in the appropriate number, and are stored in such way that the analysis required is not compromised. If quantitative soil analyses (kg/ha) are required, then soil bulk density must also be measured and this requires considerable care. [14]

13 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. PulseAustralia Limited.

14 Soilquality.org. Soil sampling for soil quality – South Australia. http://www.soilquality.org.au/factsheets/soil-sampling-for-soil-quality-south-australia

Photo 2: *To be meaningful, soil sampling needs to take into account spatial variation in the soil condition. Differences in soil type, nutrient status and other soil properties may be exhibited within a paddock.*

Source: Soilquality.org.

Accurate soil tests allow landholders to maximise the health of their soils and make sound decisions about fertiliser management to ensure crops and pastures are as productive as possible.

Identifying potential soil limitations enables landholders to develop an action plan (such as an appropriate fertiliser program) to reduce the potential of 'problem' paddocks.

Soil health looks at all aspects of the soil together, including the physical, chemical and biological components, rather than each component separately.

Healthy soil requires a balance between inputs and outputs and regular soil tests provide a valuable monitoring tool to keep an eye on soil nutrient levels and other key characteristics such as pH and salinity.

A soil imbalance can affect the ability of plants to absorb any applied fertiliser, wasting time and money. A combination of soil tests, on-farm observations, and historical records will assist in determining soil health.

Soil testing is a useful tool to provide information to support decisions about fertiliser application. However, it is important to combine test results with other information, such as specific crop or pasture requirements, available funds and fertiliser cost, methods of application, and potential income from the crop or pasture being grown.

Accurate results

Soil test results are only as accurate as the samples taken from the paddock and how they are handled leading up to laboratory analysis. If the samples do not truly reflect the soils in a certain paddock, the test results are likely to suggest an inaccurate picture of soil fertility.

Sampling not only depends on how the sample is taken but when and where.

There are four main steps in soil sampling:

- collecting representative samples
- laboratory analysis
- interpretation of test results on which to make decisions
- recording the results and actions taken for future reference.

Taking the test

There is more to soil testing than analysing the soil's nutrient status. The process incorporates the sampling procedure, soil analysis and interpretation of the results leading to a sound recommendation.

Before collecting soil samples, consult a local agronomist to discuss the need for additional tests such as deep soil nitrogen tests. In most soils the nutrients are concentrated in the top 10 cm of soil, so ensure samples are consistently taken to this depth.

When to collect samples

Changes in soil moisture, plant growth stage, and decomposition of organic matter all affect soil nutrient levels. For example, available nutrients can be low in soil samples collected during spring as nutrients are still in the plant and are not returned to the soil until after decomposition.

Check with your local agronomist as to the best time to collect soil samples in your area. Most comparisons are based on mid–summer (January to March) sampling when the soil is dry. For mobile nutrients like nitrate or potassium, deeper sampling may be required on the more sandy soils. Sampling to the rooting depth of a crop of interest might be useful for these nutrients or when studying water availability; otherwise it is generally too onerous. When assessing soil carbon stocks for accounting or budgeting purposes, a sampling depth of 30 cm is required to conform with standard accounting procedures. When sampling below 10 cm, soil samples are usually stratified by depth increments (e.g. 0-10, 10-20, 20-30 cm), depending on the objectives. When characterising a soil for the first time, sampling corresponding to the different soil layer depths (horizons) is often useful. Plant litter on the soil surface is not usually included in soil samples while plant root material is usually included, although generally sieved out prior to analysis.

Regular tests build better profile

Because many factors influence soil test results, soil analysis for one season is not conclusive. Subtle differences in soil type can impact significantly on the availability and exchange of nutrients between the soil and plants, so it is important to test soil regularly.

Testing the soil at the same time each year improves the comparison of results between years and builds a clear profile of soil health over time.

It is also important to send samples to the same laboratory for testing as results between laboratories cannot be compared easily.

Collect enough samples to make up a representative picture of a paddock. It is better to over-sample, as this will provide a more accurate picture of the soil and will help reduce unnecessary fertiliser application.

Selecting your samples

Sampling often limits the success of soil testing. One hectare of soil to a depth of 10 cm contains about 1300 t of soil. A 10 g subsample sent to a laboratory represents only one part in 1300 million. So ensure your samples are representative.

To increase test result accuracy, avoid sampling soil near fences, trees, troughs, headlands, dams, stock tracks and clumps of manure, fertiliser dumps, fertiliser bands from the previous year, burnt heaps, areas of abnormally good or poor growth or poorly drained areas.

Also avoid collecting samples from areas where fertiliser, gypsum, or lime have been applied during the preceding three months. Wet soil can alter test results due to microbial activity and mineralisation.

Account for variability

Variability of soil is often overlooked. Even individual paddocks often have variations in soil surface depth, soil type, and nutrient levels, which can be significant over relatively short distances.

Many soil types can be found in a single paddock. This, combined with management practices, can lead to varying nutrient levels within and across paddocks. Even if the paddock has a uniform single soil type, stock can spread nutrients unevenly through urine and dung. Management can concentrate or spread nutrients through clearing, burning, grazing or hay production.

Where soil differences within a paddock are obvious and areas can be treated differently, take separate samples from each area.

Where there is more than one soil type, take about 20 cores from each major soil type. Ensure each soil type is sampled and labelled separately to allow for individual analysis.

To increase productivity on larger properties, it is worthwhile classifying the land and soil types and ensuring samples are only collected from within a specific land and soil type.

Sampling sites

Take samples from across a paddock using a dedicated soil sampling tube or 'pogo'. Take at least five (preferably more) samples per hectare, covering the entire area.

Keep in mind that a hectare is 100 m by 100 m and to take five samples diagonally will involve taking samples about 30 m apart in a zig-zag pattern.

If the paddock is predominantly of one soil type take at least 40 cores, each to a depth of 10 cm. For each soil type, bulk all samples together, thoroughly mix and take a 500 g subsample to be sent to the laboratory with clear labels.

Note in your records the pattern that you used to collect samples. Following the same pattern in future years will provide a clearer picture of soil fertility trends.

Handle with care

Collect cores in a clean plastic bag and label clearly. Do not use second-hand containers or touch soil samples with bare hands, as this will contaminate the sample and affect the test results.

Air dry samples by leaving the top of the bag open to the air if there is a delay between sampling and posting. Send samples to the laboratory early in the week if possible to avoid postal delays over weekends.

Prepared soil sampling kits are available from most rural supply stores. If using an off-the shelf kit, read the instructions carefully as they may have specific instructions.

Interpreting the results

A number of laboratories are available to test and analyse your soil samples. Some services offer recommendations relating to the test results.

Contact your local agricultural consultant, agronomist, or rural supply store for the contact details of available soil testing and support services in your region.

How to take a soil sample

To obtain an accurate soil analysis, the sample cores need to be taken correctly. Before taking samples, plan how many samples are required and from where they will be collected throughout the paddock.

Ensure the stainless steel sampling tube and collection bags (use new bags for each sample) are clean before taking samples, including inside the steel tube. If using oils on clay soils, ensure they are free of nitrates and carbon.

Most soil samples are taken from the top 100 mm of surface soil. Adjustable soil sampling probes often will have marks at 100 mm intervals.

If no mark exists, set the depth stop at 100 mm using a ruler. It is vital all samples are taken from the same depth.

For large paddocks, plan to take at least 25 cores in a grid or zig-zag pattern as shown. Paddocks of more than 50 ha will require more core samples (minimum of 30 cores).

If there are two or three distinct soil types in a paddock of more than 100 ha, treat them as separate paddocks. It is recommended to take 4–8 samples within a paddock (each sample being at least 12–15 cores), each taken from within a land management unit in the paddock.

Before sampling, remove any debris from the soil surface, without disturbing the soil. Do not scuff away any plant material from the surface as this will lead to a loss of surface soil. Push in the sample tube straight until the depth stop contacts the ground.

Half-turn the sample tube and then remove it from the ground, taking care not to lose any soil from the end of the tube.

In light soils, the whole tube might need to be pushed toward the ground and a finger placed over the end of the core while the tube is parallel to the ground to ensure the soil does not fall out.

Place a sample bag over the upper end of the tube and invert as shown, emptying the core into the bag. Tap the tube with the palm of a hand to loosen the core if required.

Soil tests can be a great tool in determining the health of your soils and in turn, maximise their productivity. [15]

MORE INFORMATION

https://grdc.com.au/resources-and-publications/groundcover/ground-cover-supplements/ground-cover-issue-58-precision-agriculture-supplement/understanding-and-managing-differences-in-paddock-productivity

1.2 Paddock rotation and history

1.2.1 Break cropping

- A break crop is any crop sown to provide diversity to help reduce disease, weed, and pest levels in a paddock.
- Choice of break crop type is determined by soil type and regional climate; crop sequence is determined by market and agronomic factors.
- Sourcing regional information from research organisations, agronomists, consultants, other farmers, and industry bodies is essential when selecting the most suitable crop type and varieties.

Break crops generally refer to a pulse or oilseed crop grown instead of cereals. The decision not to grow wheat but to grow and choose a break crop is based on many factors including the relative profitability of the crops—yield by price, the cereal disease pressure, herbicide resistance and personal preference.

15 DAFWA (2016). Soil sampling and testing on a small property. https://www.agric.wa.gov.au/soil-productivity/soil-sampling-and-testing-small-property?page=0%2C2

IN FOCUS

Break cropping in the southern region

The Low rainfall crop sequencing project (LRCSP) is a collaboration between SARDI and five farming systems groups in the southern region to deliver five crop sequencing trials in the low rainfall zone of south-eastern Australia:

- Eyre Peninsula Research Foundation (EPARF) Site location: Minnipa, SA
- Upper North Farming Systems (UNFS) Site location: Appila, SA
- Mallee Sustainable Farming (MSF) Site location: Mildura, Vic
- Birchip Cropping Group (BCG) Site location: Chinkapook, Vic
- Central West Farming Systems (CWFS) Site location: Condobolin, NSW

Take home messages:

- Combined analysis of trials hosted by BCG and MSF show that first year effects of legumes are generally more reliable than oilseeds for improving subsequent wheat yield.
- Increased nitrogen supply could be measured up to two years following the break and played a key role in the break effects at Karoonda and Hopetoun where weed burden was low.
- Disease breaks tended to only last for one wheat growing season.
- Sites with a high grass weed burden require a two year break to reduce the weed seed bank to a level that enables consistent improvements in cereal production.
- Measuring the gross margin of production over a three–four year period showed that the inclusion of legume and canola breaks in the sequence was at least as profitable as continuous wheat.
- Two-year breaks can increase the variability in gross margins in the years breaks are implemented, but significant profit gains compared to continuous wheat were made at both sites for a small selection of treatments.

One year breaks trialled at Mildura showed benefits of 0.3 t/ha of extra wheat production following a legume or fallow and 0.1 t/ha following a brassica, but weed populations constrained cereal yields following a single-year break treatments.

Inclusion of most break options in the sequence was at least as profitable as continuous wheat. There were several examples where the inclusion of breaks in the sequence resulted in substantial potential profit gains. [16,17]

MORE INFORMATION

[Break crop benefits in temperate wheat production.]

[The value of break crops in low rainfall farming systems and which ones perform best.]

[Achieving success with break crops in the Mallee farming system]

PODCAST

[Break crops prove their value in low rainfall zone.]

16 McBeath T, Gupta V, Llewellyn R, Davoren B, Hunt J, Lawes R, Browne C, Ferrier D, Moodie M. 2014. GRDC Update Papers - Achieving success with break crops in the Mallee farming system https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/07/Achieving-success-with-break-crops-in-the-Mallee-farming-system

17 Moodie M, Wilhelm N. (2016). GRDC Update Papers: The value of break crops in low rainfall farming systems and which ones perform best. https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/02/The-value-of-break-crops-in-low-rainfall-farming-systems-and-which-ones-perform-the-best

1.2.2 Chickpeas as a rotation crop

Chickpea is a winter pulse crop well-suited to the southern region in rotation with cereal and canola crops. Determining the most suitable cereal–pulse–oilseed rotation requires careful planning. There are no set rules and it is best to plan a separate rotation for each cropping paddock.

The major aim should be to achieve sustainability and the highest possible overall profit, but to achieve this, the rotation must be flexible enough to cope with key management strategies such as maintaining soil fertility and structure, controlling crop diseases, and controlling weeds and their seed-set (Table 3). The same pulse should not be grown in succession, and extreme care must be taken if growing the same crop in the same paddock without a spell of at least three years. For disease management, it is recommended to aim for a break of at least four years between chickpea crops. [18] Successive cropping with the same pulse is likely to result in a rapid build-up of root and foliar diseases as well as weeds. Where possible, alternate the type of pulse crop being grown in a continuous rotation with cereals. Following dun field peas or faba beans, leave two years before sowing chickpeas. It is almost impossible to grade volunteer peas out of chickpeas.

Table 3: *Advantages and disadvantages of including chickpeas as a crop rotation.*

Advantages	Disadvantages
Chickpea is a break crop that can be used successfully in rotations to effectively break the lifecycle of cereal root diseases like take-all, cereal cyst nematode and crown rot. Chickpea plants fix their own nitrogen. They have an extensive and deep root system. Chickpea can be sown relatively late compared to wheat, which can spread the demand for machinery and labour. Chickpeas can improve soil friability. Can expand weed-control options They don't require much additional equipment. Can be sown as an opportunity crop if seasonal conditions (full profile after summer rain) allow in lower rainfall districts. Assist in snail control as chickpeas are not attractive for snail multiplication.	Chickpea is less competitive against weeds than some other crops. Crop topping to prevent weed seed set is not recommended in chickpea due to later maturity than the weeds. Chickpea is not known to break up hard pans or compacted layers in the soil profile. Nematodes can be a drawback to planting chickpeas before wheat where *Pratylenchus* species (RLN) populations are high.

In most situations, chickpeas can increase soil N by up to 35 kg nitrate-N/ha and yields of following wheat crops by up to 1 t/ha, with an additional 1% of protein. Well-grown chickpea crops have been found to contribute up to 51 kg N/ha to the subsequent cereal crop. [19] In one study, the benefit of chickpea was equivalent to the application of 60 kgN/ha as fertiliser. [20]

Wheat yields and protein in the past have tended to be poorer after chickpea crops than after other pulses. It is often better to follow chickpea with barley rather than wheat. While older chickpea varieties were a host for the root lesion nematode

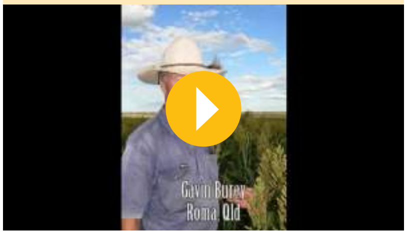
18 Pulse Australia. Southern Pulse Bulletin PA 2012 #08. Chickpea disease management strategy. http://pulseaus.com.au/storage/app/media/crops/2012_SPB-Chickpea-disease-management.pdf

19 Cox, H. W., Strong, W. M., Lack, D. W., & Kelly, R. M. PROFITABLE DOUBLE-CROPPING ROTATIONS INVOLVING CEREALS AND PULSES IN CENTRAL QUEENSLAND.

20 Marcellos, H., Felton, W. L., & Herridge, D. F. (1993). Crop productivity in a chickpea-wheat rotation. In *Proceedings 7th Australian Agronomy Conference* (pp. 276-278).

VIDEOS

8. Over the Fence: Chickpea redeemed as 'soil conditioner'.



(*Pratylenchus neglectus, and P. thornei*), newer varieties are not as susceptible to root lesion nematode multiplication. Any potential yield loss in the cereal following chickpeas can be remedied by applying an additional 10–20 kg of nitrogen fertiliser to that cereal. [21]

The disadvantages of including pulses in the rotation are possible soil erosion losses due to the lower stubble levels produced and the chance of a greater volatility in prices associated with pulses. [22]

NOTE: Do not sow chickpea in field pea or faba bean stubble. Do not sow for two years after a dun field pea type or after faba bean. It is almost impossible to grade volunteer peas out. [23]

1.2.3 Pulse effects on cereal yield

Pulses and cereal crops are complementary in a cropping rotation. The means by which a crop affects following crops include well-recognised processes related to disease, weeds, rhizosphere microorganisms, herbicide residues, residual soil water, and mineral N. They may also include two recently discovered processes. One is growth stimulation following hydrogen gas released into the soil by the legume–rhizobia symbiosis. The other is a drain on assimilates when the roots are strongly colonised by the hyphae of arbuscular mycorrhizal fungi (AMF) built up by a previous colonised host crop.

Pulses fix their own N, leaving available N in the soil for the following cereal crop. Pulses also play a vital role in helping manage major cereal root diseases (chickpea is a poor host to crown rot, cereal cyst nematode and take-all) by allowing more time for the cereal stubble to break down between host crops.

The combination of higher soil N and reduced root diseases is cumulative and can result in a dramatic increase in subsequent cereal yields. However, it is important to remember that the benefits of N fixation from pulses is not guaranteed. The amount of N fixed is determined by how well the pulse crop grows, reflecting the effectiveness of nodulation, seasonal conditions, crop management, and the level of nitrate in the soil at sowing. Soil nitrate suppresses nodulation and N fixation; hence, high soil nitrate means low N fixation (Figure 3).

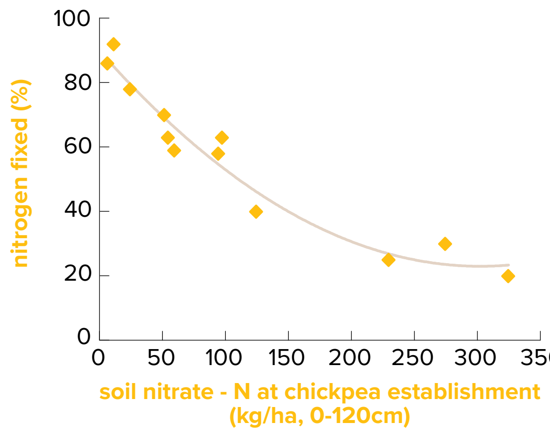


Figure 3: *Effect of soil nitrate nitrogen on nitrogen fixation by chickpea*

Source: J.A. Doughton *et al* 1993

21 Pulse Australia. Chickpea production: Southern and Western Region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

22 Cox, H. W., Strong, W. M., Lack, D. W., & Kelly, R. M. PROFITABLE DOUBLE-CROPPING ROTATIONS INVOLVING CEREALS AND PULSES IN CENTRAL QUEENSLAND.

23 Pulse Australia. Chickpeas in South Australia and Victoria. http://www.pulseaus.com.au/storage/app/media/crops/2007_Chickpeas-SA-Vic.pdf

IN FOCUS

This paper outlines the nitrogen benefits of lupins, field pea, and chickpea to wheat production in south-eastern Australia.

The research showed chickpea was second to lupin in fixing soil N.

Nitrogen balances of narrow leaf lupin (*Lupinus angustifolius* L.), albus lupin (*L. albus* L.), field pea (*Pisum sativum* L.), chickpea (*Cicer arietinum* L.), and barley (*Hordeum vulgare* L.) sown over a range of dates were examined in 1992 in a rotation study.

Yields, N_2 fixation, and crop residue N balances of the legumes were markedly influenced by sowing time, and superior performance of lupins over other species was related to higher biomass production and proportional dependence on N_2 fixation, together with a poorer harvest index.

Net soil N balance based solely on aboveground returns of N of legumes in 1992 through to harvest of wheat in 1993 was least for narrow leaf lupin-wheat (–20 kg N/ha), followed by albus lupin-wheat (–44), chickpea-wheat (–74), and field pea-wheat (–96). Corresponding combined grain N yields (legume+wheat) from the four rotations were 269, 361, 178, and 229 kg N/ha, respectively. The barley-wheat rotation yielded a similarly computed soil N deficit of 67 kg/ha. Data are discussed in relation to other studies on legume-based rotations. [24]

MORE INFORMATION

Armstrong, E. L., Heenan, D. P., Pate, J. S., & Unkovich, M. J. (1997). Nitrogen benefits of lupins, field pea, and chickpea to wheat production in south-eastern Australia. *Australian journal of agricultural research*.

Nitrate – N benefit for following cereals

The nitrate-N benefit from chickpea over a range of grain yields has been calculated from trials shown in Table 4. The terminology is important to an understanding of N budgets for chickpea and faba bean:

- 'Total N fixed'— the N fixed in both aboveground (shoots) and belowground (roots and nodules) biomass. With chickpea, 50% of total crop N is below ground.
- 'Nitrogen balance'— the difference between N inputs to the pulse crop (N fixation + N applied) and N outputs (N harvested in grain or hay + N lost (volatilised) from the crop and soil).
- 'Nitrate-N benefit'— the extra nitrate-N available at sowing in soil that grew a pulse crop in the previous season, compared with soil that grew a cereal crop.
- 'Harvest index' (HI) — for different crops, the relationship between shoot dry matter and grain yield (i.e. HI) may vary according to season and management.

By understanding the development and measurement of crop biomass and the factors that influence HI, better N and rotation management decisions can be made.

24 Armstrong, E. L., Heenan, D. P., Pate, J. S., & Unkovich, M. J. (1997). Nitrogen benefits of lupins, field pea, and chickpea to wheat production in south-eastern Australia. *Australian journal of agricultural research*.

MORE INFORMATION

Impact of crop rotations on profit, nitrogen and ryegrass seed bank in crop sequences in southern NSW.

Table 4: *Nitrate-N benefit from chickpea, over a range of grain yields (all values are kg/ha).*

Grain yield (t/ha)	Shoot dry matter (t/ha)	Low soil nitrate at sowing (50 kg N/ha)			Mod soil nitrate at sowing (100 kg N/ha)		
		N fixed	N balance	Nitrate-N benefit	N fixed	N balance	Nitrate-N benefit
1.0	2.4	31	–3	16	13	–21	4
1.5	3.6	74	22	28	47	–5	13
2.0	4.8	120	49	44	84	12	24
2.5	6.0	157	66	48	111	21	38
3.0	7.1	198	88	52	141	31	52
3.5	8.3	231	102	57	164	35	64
4.0	9.6	264	116	61	188	39	69

Source: Grain Legume Handbook (2008).

1.3 Understanding soils and pulse crop constraints

If poor crop growth and yield are occurring in a cropping paddock, or patches in it, despite good rainfall and soil moisture, a determination of what is constraining growth is needed, whatever the crop type (Photo 3).

Photo 3: *Aerial shot of chickpea crops showing wide-scale crop loss due to sodic/ saline conditions.*

Understanding growth constraints will influence crop choice or its management. Constraints may be soil-related or biological (e.g. disease, an insect pest, or a nematode). Some guidelines are provided in Table 5 and 6 below to assist in testing and diagnosis.

Table 5: *Indicative signs and likely causes of constraints to plant growth.*

Likely constraint	Indicative signs of a constraint	Possible solution
Biological	Roots may show dark lesions, knotting or discoloration (e.g. honey or brown coloured)	Identify the problem. Use crop rotations and farm hygiene and grow more resistant crops or varieties. Use fungicide or insecticide seed treatment, appropriate disease or pest control. Encourage the build-up of beneficial organisms through supplying organic substrate (e.g. stubble retention). Use direct drilling or no-till
Nutrient deficiency	Leaves or stems show characteristic symptoms of nutrient imbalance	Identify the nutrient disorder (soil or plant test). Apply appropriate fertiliser as granular, liquid injection or foliar application. Improve agronomy practices to build a healthier soil
Soil surface sodicity	Soil surface shows waterlogging, hard setting or crusting. Water ponds for several days after rain	Applying gypsum can improve soil surface sodicity by flocculating soil and so improving infiltration and exchange of sodium for calcium
Physical	Roots are deformed or may grow at a right angle. Rooting depth is restricted by presence of stones or rock, by a dense clay layer, hardpan, a plough layer or traffic compaction	Deep ripping may benefit some hardpans or compacted layers. Some ameliorant may need to be incorporated at the same time (e.g. organic matter, gypsum, lime). Controlled traffic will be needed afterwards. Growing plants with a taproot that is deep rooting can help
Chemical	There is an absence of fresh roots in the rooting zone (e.g. top 1 m of soil). The subsoil remains wet after a dry finish	Salinity: avoid sensitive crops such as chickpea and lentil, and grow more tolerant crops and varieties. If subsoil drainage is improved, then this can help to leach salts from the upper soil layers Acidity: use lime to as an ameliorant on acidic soils Sodicity: apply gypsum Alkalinity: elemental sulfur can help acidify highly alkaline soils, but large quantities will be required on heavy clay soils
Subsoil sodicity	Subsoil is lacking drainage. Structure of subsoil is coarse or dense	Sodicity: apply high rates of gypsum, but incorporation is needed, otherwise adequate rainfall and time are needed for gypsum to be effective in subsoils

Source: Grain Legume Handbook (2008).

Table 6: *Testing and decision process to follow in determining which soil constraints apply.*

Electrical conductivity (EC, 1:5 water) (dS/m)					
Check soil for EC in surface and subsoil					
Low EC <0.3 dS/m in top 10 cm Low EC <0.7 dS/m in subsoil Plant growth is not affected by salinity:			High EC >0.3 dS/m in top 10 cm High EC >0.7 dS/m in subsoil Plant growth is affected by salinity:		
Check soil for exchangeable sodium percentage (ESP) and/or dispersion			**Check soil for sodium and chloride concentration**		
No dispersion (ESP <6)		Dispersion (ESP >6)	Cl >300 mg/kg in top 10 cm soil Cl >600 mg/kg in subsoil	Cl <300 mg/kg in top 10 cm soil Cl <600 mg/kg in subsoil	
Check soil pH **(1:5 soil:water)**				**Check for gypsum crystals and sulfur concentration**	
pH <5.5	pH >8.0			S >100 mg/kg	S <100 mg/kg
Acidity constraint	**Alkalinity constraint**				
		Sodicity constraint			
			Osmotic effect due to high salt and Na/Cl toxicity,		
				High EC due to gypsum; no constraint to crop growth	**No gypsum; other salts are causing the problem**

Source: Qld Natural Resources and Water Bulletin.

For more information on soil constraints on chickpeas, see Section 14: Environmental issues.

1.4 Fallow weed control

Chickpeas are slow to emerge and initially grow slowly. They are notoriously poor competitors with weeds. Even moderate weed infestation can result in severe yield losses and harvesting problems. The best form of weed control is rotation and careful selection of paddocks largely free from winter weeds (Photo 4).

Photo 4: *Spraying weeds when small is the key to effective long fallow.*

Source: AGRONOMO.

Paddocks generally have multiple weed species present at the same time, making weed control decisions more difficult and often involving a compromise after assessment of the prevalence of key weed species. Knowledge of your paddock and early control of weeds are important for good control of fallow weeds. Information is included for the most common of the problem weeds; however, for advice on individual paddocks you should contact your agronomist.

Benefits of fallow weed control are significant:

- Conservation of summer rain and fallow moisture (this can include moisture stored from last winter or the summer before in a long fallow) is integral to winter cropping in the southern region, particularly so as the climate moves towards summer-dominant rainfall.
- Modelling studies show that the highest return on investment in summer weed control is for lighter soils, or where soil water is present that would support continued weed growth.

Trials exploring methods for summer grass control have found:

- Glyphosate-resistant and tolerant weeds (including annual ryegrass) are a major threat to our reduced-tillage cropping systems.
- Although residual herbicides will limit re-cropping options and will not provide complete control, they are key to successful fallow management.
- Double-knock herbicide strategies (sequential application of two different weed control tactics) are useful tools but the herbicide choices and optimal timings will vary with weed species.
- Other weed management tactics can be incorporated, e.g. crop competition, to assist herbicide control.
- Cultivation may need to be considered as a salvage option to avoid seed-bank salvage. [25]

Stopping weed growth in the fallow can lead to yield increases in the following crop via several pathways. These include:

- Increased plant-available water
- A wider and more reliable sowing window
- Higher levels of plant-available N

25 GRDC (2012) Make summer weed control a priority—Southern region. Summer Fallow Management, GRDC Fact Sheet January 2012, https://www.grdc.com.au/~/media/8F16BE33A0DC4460B17317AA266F3FF4.pdf

VIDEOS

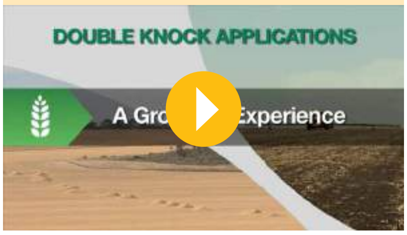
9. GCTV5: Managing summer fallow.

MORE INFORMATION

Summer weed fallow management: a reference manual for grain growers and advisers in the southern and western grains regions of Australia.

VIDEOS

10. Double knock application – a Grower's Experience.



- Reduced levels of weed-vectored diseases and nematodes
- Reduced levels of rust inoculum via interruption of the green bridge
- Reduced levels of diseases vectored by aphids that build in numbers on summer weeds, and
- Reduced weed physical impacts on crop establishment. [26]

1.4.1 Management strategies

How farming country is managed in the months or years before sowing can be more important in lifting water use efficiency (WUE) than in-crop management. Of particularly high impact are strategies that increase soil capture and storage of fallow rainfall to improve crop reliability and yield.

Practices such as controlled traffic farming and long term no-till seek to change the very nature of soil structure to improve infiltration rates and improve plant access to stored water by removal of compaction zones.

Shorter term management decisions can have an equal or even greater impact on how much plant-available water (PAW) is stored at sowing. These include decisions such as crop sequence/rotation that dictate the length of the fallow and amount of stubble cover, how effectively fallow weeds are managed, stubble management, and decisions to till/not to till at critical times.

While many factors influence how much plant available water is stored in a fallow period, good weed management consistently has the greatest impact. [27]

Double-knock strategies

Double-knock refers to the sequential application of two different weed-control tactics applied in such a way that the second tactic controls any survivors of the first. Most commonly used for pre-sowing weed control, this concept can also be applied in-crop. [28]

Consider the species present, interval timing, and water rate. Double-knock herbicide strategies are useful tools for managing difficult-to-control weeds, but there is no 'one size fits all' treatment. The interval between double-knock applications is a major management issue for growers and contractors. Shorter intervals can be consistently used for weeds where herbicides appear to be translocated rapidly or when growing conditions are very favourable. Longer intervals are needed for weeds where translocation appears slower. Critical factors for successful double-knock approaches are for the first application to be on small weeds and to ensure good coverage and adequate water volumes, particularly when using products containing paraquat. Double-knock strategies are not fail-proof and are rarely effective for salvage weed-control situations unless environmental conditions are exceptionally favourable.

Herbicide application

Broadleaf weeds and herbicide resistant grasses can cause major problems and a careful management strategy must be worked out well in advance. If possible, control weeds in the year prior to sowing chickpea or avoid paddocks with specific weeds that cannot be controlled by the available herbicides. [29]

Herbicide options for broad-leaved weed control are very limited. Growers will need to consider alternative control strategies if the standard treatment of post-sow pre-emergent Simazine is unlikely to provide adequate control, i.e.:

- The use of Balance®

26 GRDC. (2014). Summer fallow weed management. https://grdc.com.au/Resources/Publications/2014/05/Summer-fallow-weed-management

27 GRDC. (2014). Summer fallow weed management. https://grdc.com.au/Resources/Publications/2014/05/Summer-fallow-weed-management

28 C Borger, V Stewart, A Storrie. Double knockdown or 'double knock'. Department of Agriculture and Food Western Australia, http://www.agric.wa.gov.au/objtwr/imported_assets/content/pw/weed/iwm/tactic%202.2doubleknock.pdf

29 Pulse Australia. Chickpea production: Southern and Western Region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

- Use of trifluralin
- Inter-row cultivation is only an option in wide row systems
- Inter-row shielded sprayer (glyphosate) is only an option in wide row systems
- Post emergent Broadstrike® may be damaging (refer to label)
- Directed post-emergence sprays of Broadstrike® and/or Simazine

Herbicide options for grassy weed may be very limited if herbicide resistant ryegrass is present. Growers will need to consider alternative control strategies if the standard trifluralin pre-sowing treatment and post-sow pre-emergent Simazine is unlikely to provide adequate control i.e.:

- The use of Balance® or newer grass herbicides such as Boxer Gold® and Sakura®.
- Use of group A herbicides post-emergent if herbicide resistance not present
- Inter-row cultivation is only an option in wide row systems
- Inter-row shielded sprayer (glyphosate) is only an option in wide row systems
- Crop topping or weed wiping are not options to prevent seed set of escape weeds.

Avoid paddocks with high seed numbers of herbicide resistance unless a programmed strategy is in place. [30]

Newer grass herbicides are now available, including Boxer Gold, Sakura. See www.apvma.gov for label information.

For more information on weed management, see Section 6: Weed Control.

VIDEOS

11. Burning for weed and snail control.

1.5 Fallow chemical plant-back effects

Plant-back periods are the obligatory times between the herbicide spraying date and safe planting date of a subsequent crop. Some herbicides have a long residual persistence. The residual is not the same as the half-life. Although the amount of chemical in the soil may break down rapidly to half the original amount, what remains can persist for long periods, e.g. sulfonylureas (chlorsulfuron). The rate of decay is influenced by soil pH and moisture levels.

Herbicides with long residuals can affect subsequent crops, especially if they are effective at low levels of active ingredient, such as the sulfonylureas. On labels, this will be shown by plant-back periods, which are usually listed under a separate plant-back heading or under the 'Protection of crops etc.' heading in the 'General Instructions' section of the label. [31]

MORE INFORMATION

Herbicide residues in pulses

In wheat-chickpea rotations, adhere to label plant-back restrictions. Broadstrike®, Eclipse®, Lontrel®, metsulfuron are used in preceding crops in the Southern region, subject to plant-back time and rainfall requirements.

The use of long-term residual SU herbicides such as Monza®, chlorsulfuron (Glean®, Lusta®), and Logran® in wheat should be avoided when re-cropping to chickpeas.

Rotational crop plant-back intervals for southern Australia

Where areas have received limited rain during the spring and summer months, there is potential for herbicide residues to still be present in the soil when sowing commences in autumn, unless there are mild temperatures and adequate moisture at least a month or more before sowing (Table 7).

Conditions required for breakdown

Warm, moist soils are required to break down most herbicides through the processes of microbial activity. For the soil microbes to be most active, they need good moisture and an optimum soil temperature range of 18°C to 30°C. Extreme temperatures above

30 Pulse Australia. Southern Pulse Bulletin PA 2010 #05 – Chickpea checklist for southern growers. http://www.pulseaus.com.au/storage/app/media/crops/2010_SPB-Chickpea-checklist-south.pdf

31 B Haskins (2012) Using pre-emergent herbicides in conservation farming systems. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0003/431247/Using-pre-emergent-herbicides-in-conservation-farming-systems.pdf

or below this range can adversely affect soil microbial activity and slow herbicide breakdown. Very dry soil also reduces breakdown. To make matters worse, where the soil profile is very dry it requires a lot of rain to maintain topsoil moisture for the microbes to be active for any length of time.

Risks

In those areas that do not experience conditions which will allow breakdown of residues until just prior to sowing, it is best to avoid planting a crop that is sensitive to the residues potentially present on the paddock, and opt for a crop that will not be affected by the suspected residues. In most cases, cereals or canola would be better options as these crops are comparatively less affected by herbicide residues. If dry areas do get rain and the temperatures become milder, then they are likely to need substantial rain (more than the label requirement) to wet the sub-soil, so the topsoil can remain moist for a week or more. This allows the microbes to be active in the top-soil where most of the herbicide residues will be found. Sensitive crops include legume pastures (e.g. clovers, Lucerne, or forage legumes) and pulse crops (eg. lentils, lupins, fieldpeas, faba beans, or vetch). [32]

MORE INFORMATION

Rotational crop plant-back intervals for southern Australia.

Table 7: *Minimum re-cropping intervals and guidelines for common broadacre herbicides.*

Product	Rate	Plant-back period	Wheat	Barley	Oats	Canola	Legume Pasture	Pulse Crops
2,4-D Ester 680 *	0–510 ml/ha	(days)	1	1	1	14	7	7
	510–1,150 ml/ha		3	3	3	21	7	14
	1,150–1,590 ml/ha		7	7	7	28	10	21
Amicide Advance 700® *	0–500 ml/ha	(days)	1	1	1	14	7	7
	500–980 ml/ha		3	3	3	21	7	14
	980–1,500 ml/ha		7	7	7	28	10	21
Kamba® 500 *	200 ml/ha	(days)	1	1	1	7	7	7
	280 ml/ha		7	7	7	10	14	14
	560 ml/ha		14	14	14	14	21	21
Hammer 400® EC					No residual effects			
Nail 420 EC®					No residual effects			
Goal®					No residual effects			
Striker®					No residual effects			
Sharpen®	26 g/ha	(weeks)	-	-	-	16	-	-
Lontrel®	300 ml/ha	(weeks)	1	1	1	1	36	36
Garlon 600®		(weeks)	1	1	NS	NS	NS	NS
Allyv **		(weeks)	2	6	36	36	36	36
Logran® #		(months)	-	-	-	12	12	12
Gleanv **		(months)	-	9	6	12	12	12
Grazon Extra®/Grazon DS®		(months)	9	9	NS	9	24	24
Tordon 75D®, Tordon 242®		(months)	2	2	NS	4	9	6
Tordon Fallow Boss®		(months)	9	9	NS	12	20	20

* 15 mm rainfall required to commence plant-back period
** Period may extend where soil pH is greater than 7
# Assumes 300 mm rainfall between chemical application and sowing
NS Not Specified

Source: RMS.

32 Dow AgroSciences. Rotational crop plant-back intervals for southern Australia. http://msdssearch.dow.com/PublishedLiteratureDAS/dh_0931/0901b80380931d5a.pdf?filepath=au&fromPage=GetDoc

1.5.1 Herbicide residues in soil

Residues from herbicides used in the current or previous crop could impact on subsequent crop choice in rotations (Photo 5). Crop damage could occur if this is ignored, particularly where rainfall has been minimal. Pulse and other crop types differ in their sensitivity to residual herbicides so check each herbicide used against each crop type. Check herbicide history in paddocks for any chemical that may cause any detrimental effect to chickpeas. Herbicide choice in cereal and oilseed crops may have to accommodate the planning of a pulse crop next in the rotation sequence. For example, it could be 10 months before a chickpea crop can be grown after use of an imidazolinone ('imi') herbicide, and likewise over 24 months after chlorsulfuron has been applied on high pH soils. [33]

Photo 5: *Herbicide residue affected plants appear pale and stunted.*

Source: DAFWA.

Herbicides such as group B's (Logran®, Glean®, Hussar® and Ally®) typically pose the greatest threat if they are persistent in the soil when chickpeas are planted. Typical symptoms are stunting and yellowing of the plants if affected. Other herbicides to be wary of are Lontrel™, Amine™ (used over summer) and Dicamba (used prior to

33 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

cropping). These herbicides (group I's) can cause twisting and growth distortions of plants. [34]

For more information, see Section 6: Weed control.

VIDEOS

12. GCTV16: Desi Chickpea – considering herbicide residuals.

1.6 Seedbed requirements

Preparation of a seedbed to ensure good seed soil contact is an important element in successful crop establishment. A firm, smooth seedbed with most of the previous crop residue incorporated is best. This will allow proper depth of planting as well as good seed-soil contact, which is essential for rapid germination and emergence. [35]

In other pulses (faba beans) in southern Australia, it is recommended that the previous crop stubble be heavily grazed, slashed, or burnt in order to obtain a flat seedbed. Rolling after seeding should be considered on stony or extremely cloddy soil, because pods set low on the plant may cause harvest difficulties. [36]

1.7 Soil moisture

1.7.1 Dryland

Chickpeas are deemed to require >350 mm annual rainfall, but there are opportunities to grow them in lower rainfall areas if adequate soil moisture is present at sowing (e.g. >20 mm of stored soil water at 0–60 cm depth). The best guide to assessing soil water storage is to put down several soil cores. [37]

Tillage

Photo 6: *The impact of tillage varies with the tillage implement used: inversion tillage using a mouldboard plough, as pictured here, results in greater impacts than using a chisel or disc plough.*

Source: GRDC.

Research from Qld found that one-time tillage with chisel or offset disc in long-term, no-tillage helped to control winter weeds, and slightly improved grain yields and profitability, while retaining many of the soil quality benefits of no-till farming systems (Photo 6). Tillage reduced soil moisture at most sites; however, this decrease in soil moisture did not adversely affect productivity. This could be due to good rainfall received after tillage and prior to seeding and during the crop of that year. The occurrence of rain between the tillage and sowing or immediately after sowing

34 M Witney, GRDC. Update Papers. Chickpea management and agronomy. https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2012/07/Chickpea-management-and-agronomy

35 https://hort.purdue.edu/newcrop/afcm/chickpea.html

36 Agriculture Victoria, 2013. Growing Faba Bean. http://agriculture.vic.gov.au/agriculture/grains-and-other-crops/crop-production/growing-faba-bean

37 Pulse Australia. Southern Pulse Bulletin PA 2010 #05 – Chickpea checklist for southern growers. http://www.pulseaus.com.au/storage/app/media/crops/2010_SPB-Chickpea-checklist-south.pdf

is necessary to replenish soil water lost from the seed zone. This suggests the importance of timing of tillage and of considering the seasonal forecast. Future research will determine the best timing for strategic tillage in no-till systems (Photo 7). [38] Note that these results are from one season and research is ongoing, so any impacts are likely to vary with subsequent seasonal conditions.

Photo 7: *Strategic tillage can provide control for herbicide-resistant weeds and those that continue to shed seed throughout the year. Here it has been used for control of barnyard grass in fallow.*

Source: GRDC.

Research in Spain found that conventional tillage practices led to better chickpea root development than no-tillage practices. [39]

1.7.2 Irrigation

Key points:

- Select fields with good layout and tail water drainage (Photo 8).
- Avoid high bulk density or high clay content soils that do not internally drain quickly.
- Avoid acid, saline, or sodic soils (see levels below).
- Pre-irrigate or water-up to fill the soil profile wherever possible.
- Irrigate early at 60–70% of field capacity to avoid crop stress and soil cracking open.

Full or supplementary irrigation of chickpea is common in northern Australia where chickpea is grown in rotation with other irrigated crops, such as cotton. Management requirements for irrigated chickpea are the same as for dryland, but their sensitivity to waterlogging, for even a short time, can result in severe losses, particularly if the crop is also under stress from herbicides or disease.

Using sprinkler irrigation equipment reduces the risk of waterlogging, even during flowering and pod-fill; however, there may be a higher risk of foliar disease, e.g. Botrytis grey mould and Ascochyta blight, due to the increased irrigation frequency and leaf wetness. [40]

Key tips for success

- **Drainage**: Ensure the layout allows irrigation and drainage within eight hours.

MORE INFORMATION

GRDC Strategic Tillage Tips and Tactics fact sheet.

38 Y Dang, V Rincon-Florez, C Ng, S Argent, M Bell, R Dalal, P Moody, P Schenk (2013) Tillage impact in long term no-till. GRDC Update Papers Feb. 2013, https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/02/Tillage-impact-in-long-term-no-till

39 Muñoz-Romero, V., López-Bellido, L., & López-Bellido, R. J. (2012). The effects of the tillage system on chickpea root growth. *Field Crops Research*, *128*, 76-81.

40 Pulse Australia. 2015. Chickpea Production: Southern and Western Region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

- **Soil structure**: Good soil structure ensures good water infiltration, root penetration, and internal drainage.
- **Subsoil moisture**: Pre-irrigate or water-up to achieve adequate soil moisture for uniform emergence and during the vegetative stage. Irrigate prior to flowering to ensure a good profile of moisture during flowering and pod fill.
- **Sown on time**: Sow recommended varieties within the preferred sowing window for your location.
- **Crop establishment**: Use good quality seed and germination test retained seed (Photo 9). Aim for a plant population of 35 to 40 plants per square metre.
- **Adequate nutrition**: While chickpeas are efficient at extracting soil phosphorus, it is wise to apply adequate phosphorus relative to the paddock history and soil test results. Approximately 40 kg of P per hectare is required for a 4 tonne crop. Good inoculation procedures with the appropriate rhizobium should meet the N requirements of chickpeas; however, low zinc and sulfur levels should also be addressed.
- **Soil moisture**: Check soil moisture regularly to ensure timely irrigations to avoid stress or possible crop damage. Moisture monitoring equipment is now available at reasonable prices and can assist in more precise measuring, particularly at depth. Ensure plants do not stress during the reproductive stage and have adequate available soil water for the entire growing season. [41]

Photo 8: *Poor quality seed on the left—all seed was sown on the same day.*

Source: Pulse Australia.

41 Pulse Australia. Southern Pulse bulletin PA 2010 #17 – Irrigated chickpea management. http://www.pulseaus.com.au/storage/app/media/crops/2010_SPB-Chickpea-irrigation.pdf

Photo 9: *Irrigated beds with damage from water remaining in the tail drain.*

Source: Pulse Australia.

Irrigated chickpea crops can be very profitable and rewarding with well managed crops yielding in excess of 3.5 t/ha. High yields have occurred across a wide range of soil types and irrigation layouts through a combination of correct paddock selection, precise irrigation scheduling and close attention to chickpea agronomy. In addition, chickpeas can contribute to crop rotations because of their ability to fix nitrogen and provide a disease and weed break for following cereal crops.

Full or supplementary irrigation of chickpea is common in districts where chickpea is grown in rotation with other irrigated crops, such as cotton. Management requirements for irrigated chickpea are the same as for dryland, but their sensitivity to waterlogging, for even a short time, can result in severe losses, particularly if the crop is also under stress from herbicides or disease. [42]

Irrigation requirement for chickpea has been found to coincide with flowering and seed development period. [43]

Factors to consider when planning for irrigated chickpea production include:

- Avoid heavy clay or dense soil types (bulk density >1.5) that do not drain freely and are subject to waterlogging.
- Select fields with an effective irrigation layout, such as beds or hills, and relatively good grades.
- A border-check layout that is steeper than 1:800 grade is suitable provided there are short runs on free draining soils that can be irrigated quickly and do not remain saturated.
- Rolling may be required to flatten the ridges left by press wheel furrows or to flatten clods.
- Irrigation can be used in activating and incorporating a number of pre-emergent herbicides.
- Pre-irrigate to fill the moisture profile prior to planting chickpea crops, unless there has already been sufficient rainfall. Watering up is most effective in bed,

42 Pulses Australia. Chickpea Production: Southern and Western regions. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

43 Malhotra, R. S., Singh, K. B., & Saxena, M. C. (1997). Effect of Irrigation on Winter-Sown Chickpea in a Mediterranean Environment. *Journal of Agronomy and Crop Science*, *178*(4), 237-243.

row and sprinkler systems, but is not recommended for border-check layout unless soil moisture is insufficient to achieve a uniform germination.

- As a general rule, irrigation of the emerged crop should start early when there is a deficit of between 30–40 mm and around 60–70% field capacity. Schedule irrigation using soil moisture indicators rather than the crop growth stage.
- Time irrigation application to prevent moisture stress during flowering and podding and to reduce the impact of high temperatures on yield, quality and grain size. This is particularly important with large Kabuli types. Chickpeas are very sensitive to waterlogging during flowering and podding so great care is required to provide adequate soil moisture without causing waterlogging.
- In furrow irrigation systems, water every second row to avoid waterlogging. Doubling up the number of siphons can increase water flow and reduce irrigation time.
- Aim to have watering completed in less than eight hours, and have good tail water drainage to avoid any waterlogging in the crop area.
- Avoid irrigating if rain is forecast for the near future.
- In border-check layouts and paddocks with heavy soil types or long runs: if in doubt, do not water. [44]

It is important for growers and agronomists to base yield expectations on the total water supply available. This includes a combination of the amount of soil water in the profile, likely in-crop rainfall, and irrigation water supply. A general rule of thumb for chickpeas can be based on 1 tonne grain per megalitre water supply (per hectare).

To offset the good performances, there are growers who have only achieved yields of 1.0–1.5 t/ha and some of the common causes have been:

- problems with poor crop establishment and vigour (seed quality, seedbed, herbicides)
- unsuitable soils limiting water extraction (sodic or saline subsoils)
- poor scheduling of in-crop irrigation
- restricted water supply limiting yield.

Chickpeas are very sensitive to waterlogging and even if waterlogged for a short period of time, crop losses can be severe. This has particularly occurred where crops have been moisture stressed, allowing soils to dry out to depth and often crack open (Photo 10).

Waterlogging is a stress on a chickpea crop and when combined with other stresses such as moisture stress, damaged root systems, disease, herbicide injury, or sodic and saline soils can be a disaster. Watering during flowering or podfill when the crop is more sensitive further increases the risk of yield loss. [45]

44 Pulses Australia. Chickpea Production: Southern and Western regions. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

45 Pulse Australia. Southern Pulse bulletin PA 2010 #17 – Irrigated chickpea management. http://www.pulseaus.com.au/storage/app/media/crops/2010_SPB-Chickpea-irrigation.pdf

Photo 10: *Chickpea on the left received a very late irrigation.*

Source: Pulse Australia.

Irrigation management strategy for chickpea

- Pre-irrigate to fill the moisture profile prior to planting chickpea crops, unless there has already been sufficient rainfall. Watering up is most effective in bed, row and sprinkler systems, but is not recommended for border-check layout unless soil moisture is insufficient to achieve a uniform germination. Ensure that seed placement allows at least 7 cm of soil above the seed if using Balance® or simazine and the soil surface is left flat to prevent herbicide leaching into the plant furrow.
- As a general rule, irrigation of the emerged crop should start early when there is a deficit of between 30–40 mm and around 60–70% field capacity. Schedule irrigation using soil moisture indicators rather than the crop growth stage.
- Time irrigation application to prevent moisture stress during flowering and podding and to reduce the impact of high temperatures on yield, quality and grain size. This is particularly important with large Kabuli types. Chickpeas are very sensitive to waterlogging during flowering and podding, so great care is required to provide adequate soil moisture without causing waterlogging.
- In furrow irrigation systems, water every second row to avoid waterlogging. Doubling up the number of siphons can increase water flow and reduce irrigation time.
- Aim to have watering completed in less than eight hours, and have good tail water drainage to avoid any waterlogging in the crop area.
- Avoid irrigating if rain is forecast for the near future.
- In border-check layouts and paddocks with heavy soil types or long runs: if in doubt, do not water. [46]

Sowing rate and row spacing

In southern NSW the target density is 35–45 plants/m^2. Adjust sowing rates to take account of seed size, germination percentage, and establishment conditions. In southern NSW, irrigation growers have sown at up to 90 cm row spacing on beds (Photo 11) without a significant yield penalty and has also allowed the use of shielded sprayers.

46 Pulses Australia. Chickpea Production: Southern and Western regions. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

Photo 11: *Chickpea on irrigated beds with 90 cm row spacing.*

Source: Pulse Australia.

Irrigation techniques to reduce the period of waterlogging

- For furrow irrigation, water every second row.
- Doubling up siphons to speed up water flow.
- Do not irrigate if there is likelihood of rain.
- Ensure that tail water drains away quickly.

Spray irrigation

The risk of waterlogging is significantly reduced when using lateral move or centre pivot irrigators compared to flood as the amount and timing of water application can be better controlled. However wet foliage from more frequent irrigations can increase the risk of fungal diseases—particularly Ascochyta blight and Botrytis grey mould. Greater attention to disease management, and monitoring variety chosen in relation to disease resistance is important. [47]

MORE INFORMATION

Irrigated Chickpeas – Best Practice Guide.

1.8 Yield and targets

Seed yields of chickpea are low (world average 623 kg ha-1), compared to many other crops. Early field trials indicate that seed yield in chickpea is controlled by the Harvest Index of branches and that seed yields might be raised by restricting branches in high density stands to no more than two, each with a high harvest index. [48]

Starting soil water can also have a strong influence on the yield expectation of chickpea as well as the riskiness of production.

The most accurate prediction of a variety's performance is a stable yield in many locations over several years.

Yield results from Pulse Breeding Australia (PBA) and National Variety Trials (NVT) are available from the NVT website, as well from the specific Pulse Variety Management Package (VMP) brochure. Long term yields can be represented in several different

47 Pulse Australia. Southern Pulse bulletin PA 2010 #17 – Irrigated chickpea management. http://www.pulseaus.com.au/storage/app/media/crops/2010_SPB-Chickpea-irrigation.pdf

48 K.H.M. Siddique and A.M. Sedgley. An ideotype for chickpea (*Cicer arietinum* L.) in a dry mediterranean environment. 2nd Aust. Agron Conf.©,Uni of WA.

ways but are typically displayed as either site specific, averaged over multiple years (Figure 4), or for each year averaged over multiple sites for a region. All trial sites are disease free.

Results lists are approximates for the following varieties (ordered from highest t/ha to lowest).

Figure 4: *Yield group (t/ha) for Desi Chickpea (top) and Kabuli chickpea (bottom) in the Southern region (SA, Vic) between 2012-2016. Yield Group: Presents data on half tonne yield intervals based on trials that match the yield range from across all years.*

Source: NVT Online

IN FOCUS

1.8.1 Critical period for chickpea yield

Crop species have their own characteristic 'window' of development when yield is more vulnerable to stresses such as drought, nutrient deficiency, frost and heatwaves.

To determine the critical window for chickpea yield, the South Australian Research and Development Institute (SARDI) set up trials at Roseworthy (sowing date 7 June) and Turretfield (sowing dates 14 June and 9 July) in South Australia.

Crops of PBA Boundary$^{\phi}$ and PBA Slasher$^{\phi}$ were shaded for two weeks at different stages during the growing season. Untreated controls yielded three tonnes per hectare.

The trials showed that the critical window for chickpeas starts at about 300 'degree-days' before flowering and the most vulnerable stage for yield was found to be 200 'degree-days' after flowering.

Degree-days are a calculation of time based on daily temperature and are necessary to account for the fact that crops develop faster at high temperatures. For the crop, one day at 15°C is not the same as a day at 10°C.

For example, if chickpeas are grown in conditions where the daily mean temperature is 15°C, the critical stage of 200 degree-days will be reached 13 days after flowering (200 ÷ 15 = 13). If chickpeas are grown in a warmer region or the crop was sown late with, for example, a daily mean temperature of 20°C, the crop will reach the 200-degree-days mark 10 days after flowering (200 ÷ 20 = 10).

Ensuring good growing conditions (sufficient supply of water and nutrients) and avoiding stress (such as frost and heat) during the critical window are essential for high-yielding chickpea crops. [49]

MORE INFORMATION

L Lake, V Sandras. SARDI. (Nov – Dec 2014). Ground Cover issue 113: Critical period for chickpea yield.

1.8.2 Seasonal outlook

Australia's climate, and in particular rainfall, is among the most variable on earth; consequently, crop yields vary from season to season. In order to remain profitable, crop producers must manage their agronomy, crop inputs, marketing, and finance to match each season's yield potential.

Before planting, identify the target yield in grain, hay or DM required to be profitable:

- Do a simple calculation to see how much water you need to achieve this yield.
- Know how much soil water you have (treat this water like money in the bank).
- Think about how much risk your farm can take.
- Consider how this crop fits into your cropping plan—will the longer-term benefits to the system outweigh any short-term losses?
- Avoiding a failed crop saves money now and saves stored water for future crops. [50]

Mobile applications (apps) are available for decision support, providing tools for ground-truthing precision agriculture data. Apps and mobile devices are making it easier to collect and record data on-farm. The app market for agriculture is evolving rapidly, with new apps becoming available on a regular basis. For more information, download the GRDC Update paper, Managing data on the modern farm. [51]

The Bureau of Meteorology has recently moved from a statistics-based to a physics-based (dynamical) model for its seasonal climate outlooks. The new system has better overall skill, is reliable, allows for incremental improvements in skill over time, and provides a framework for new outlook services including multi-week/monthly outlooks and the forecasting of additional climate variables. [52]

49 L Lake, V Sandras. SARDI. (Nov – Dec 2014). Ground Cover issue 113: Critical period for chickpea yield. https://grdc.com.au/Media-Centre/Ground-Cover/Ground-Cover-Issue-113-NovDec-2014/Critical-period-for-chickpea-yield

50 J Whish (2013) Impact of stored water on risk and sowing decisions in western NSW. GRDC Update Papers 23 July 2013, https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/07/Impact-of-stored-water-on-risk-and-sowing-decisions-in-western-NSW

51 http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/02/Managing-data-on-the-modern-farm

52 J Sabburg, G Allen (2013) Seasonal climate outlook improvements changes from historical to real time data. GRDC Update Papers 18 July 2013, http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/07/Seasonal-climate-outlook-improvements-changes-from-historical-to-real-time-data

Australian CliMate

Australian CliMate is a suite of climate analysis tools delivered on the web, iPhone, iPad, and iPod Touch devices. CliMate allows you to interrogate climate records to ask questions relating to rainfall, temperature, radiation, and derived variables such as heat sums, soil water and soil nitrate, as well as El Nino Southern Oscillation status. It is designed for decision makers such as farmers whose businesses rely on the weather. Download from the Apple iTunes store or Australian CliMate.

One of the CliMate tools*, Season's progress?*, uses long-term (1949 to present) weather records to assess progress of the current season (rainfall, temperature, heat sums and radiation) compared with the average and with all years. It explores the readily available weather data, compares the current season with the long-term average, and graphically presents the spread of experience from previous seasons. Crop progress and expectations are influenced by rainfall, temperature and radiation since planting. *Season's progress?* provides an objective assessment based on long-term records:

- How is the crop developing compared to previous seasons, based on heat sum?
- Is there any reason why my crop is not doing as well as usual because of below average rainfall or radiation?
- Based on the season's progress (and starting conditions from Howwet–N?), should I adjust inputs?

For inputs, *Season's progress?* asks for the weather variable to be explored (rainfall, average daily temperature, radiation, heat sum with base temperatures of 0, 5, 10, 15 and 20°C), a start month, and a duration. As outputs, text and two graphical presentations are used to show the current season in the context of the average and all years. Departures from the average are shown in a fire-risk chart as the departure from the average in units of standard deviation.

1.8.3 Fallow moisture

For a growing crop there are two sources of water: first, the water stored in the soil during the fallow, and second, the water that falls as rain while the crop is growing. As a farmer, you have some control over the stored soil water; you can measure how much you have before planting the crop. Long-range forecasts and tools such as the SOI can indicate the likelihood of the season being wet or dry; however, they cannot guarantee that rain will fall when you need it.

Crop yield is highly influenced by fallow rainfall storage and soil moisture at sowing. A high frequency of low-yielding crops with small margins may be less profitable than a well-planned rotation which includes some crops grown on long fallow.

Good farming systems take into account soil health, stubble cover, and fallow management to reduce runoff and evaporation, thereby storing as much soil moisture as possible. A small amount of extra soil stored moisture can go close to doubling crop profitability.

Extra soil water storage can double profit.

An extra 20 mm of soil stored water could add 400 kg/ha to yield—enough to double the profit in some situations! Soil moisture storage improves with soil health, which is about building soil organic matter with plenty of earthworms. This can be destroyed by tillage, compaction, or low soil cover. Controlled traffic in combination with zero tillage improves infiltration, which results in less runoff and more even moisture storage across a paddock. Good soil structure can also reduce waterlogging and its effect on crop yield and nitrogen losses. [53]

53 Fritsch S, Wylie P. GRDC Update Papers. 2015. Finding more yield and profit from your farming system. https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2015/02/Finding-more-yield-and-profit-from-your-farming-system

1.8.4 Water Use Efficiency

Water Use Efficiency (WUE) is the measure of a cropping system's capacity to convert water into plant biomass or grain (Figure 6). It includes both the use of water stored in the soil and rainfall during the growing season.

- WUE relies on:
- the soil's ability to capture and store water;
- the crop's ability to access water stored in the soil and rainfall during the season;
- the crop's ability to convert water into biomass; and
- the crop's ability to convert biomass into grain (harvest index). [54]

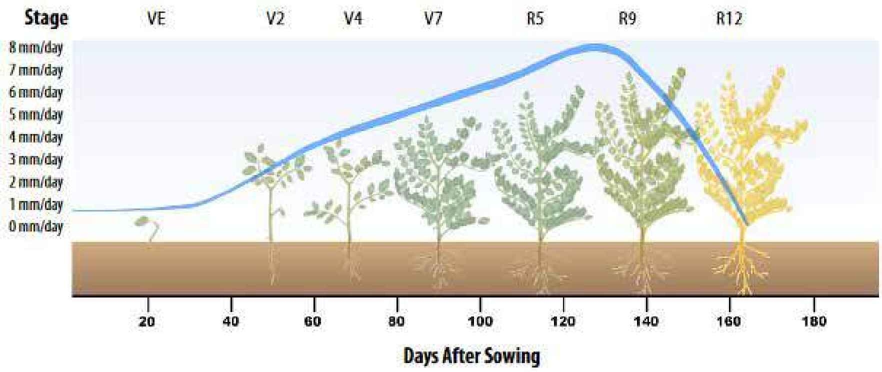


Figure 5: *Average daily water use pattern for chickpea at critical growth stages. NOTE: information based on research in the Northern region.*

Source: WATERpak.

The strongest determinant of chickpea grain yield and its water use under rainfed conditions is rainfall and its distribution. Water availability is a major constraint for production of grain in Australia and improving WUE is a primary target for growers, breeders, and agronomists. WUE benchmarks can be used to derive attainable yield for a location and season.

Seasonality and size of rainfall events also influence crop WUE. In the southern and western growing regions, rainfall is winter-dominant, falling during the crop's growing season. Soil evaporation, favoured by winter rainfall and small events, is the main unproductive source of water loss in southern and western regions. Collectively, vapour pressure deficit and rainfall patterns are the main climate determinants of location-specific WUE. [55]

Large inter-seasonal fluctuations in weather can result in larger inter-seasonal fluctuations in water use, and therefore in production of legumes. Seasonal evapotranspiration (ET) has been found to significantly correlate with seasonal rainfall for chickpeas.

Potential transpiration efficiencies (TE) of 15 kg/ha–1 mm–1 , together with soil evaporation (Es) values of 100–125 mm can be used as benchmark values to assess the yield potential of cool season grain legume crops in low rainfall Mediterranean-type environments. [56]

Research conducted in a Mediterranean-type environment investigated the effect of supplemental irrigation and sowing date on yield and WUE in winter-sown chickpea. Limited supplemental irrigation can play a major role in boosting and stabilizing the productivity of winter-sown chickpea. Four levels of supplemental irrigation (SI) were implemented as treatments: full SI, 2/3 SI, 1/3 SI, and no SI; i.e. rainfed. The results showed that chickpea yield per unit area increases with both earlier sowing

54 GRDC. (2009). Water Use Efficiency – Fact Sheet. Northern Region.

55 V Sandras, G McDonald. GRDC. (2012). Water Use Efficiency of grain crops in Australia: principles, benchmarks and management.

56 Siddique, K. H. M., Regan, K. L., Tennant, D., & Thomson, B. D. (2001). Water use and Water Use Efficiency of cool season grain legumes in low rainfall Mediterranean-type environments. *European Journal of Agronomy*, *15*(4), 267-280.

VIDEOS

13. GCTV12: Water Use Efficiency Initiative.

MORE INFORMATION

Gan, Y. T., Warkentin, T. D., Bing, D. J., Stevenson, F. C., & McDonald, C. L. (2010). Chickpea Water Use Efficiency in relation to cropping system, cultivar, soil nitrogen and Rhizobial inoculation in semiarid environments. *Agricultural water management*, 97(9), 1375-1381. *97*(9), 1375-1381.

and increased SI. However, WUE under supplemental irrigation decreases with earlier sowing, due to the relatively large increase that occurs in the amount of evapotranspiration at early sowing dates. The 2/3 Supplemental irrigation level was found to give the optimum Water Use Efficiency for chickpea. [57]

Nitrogen-deficient soils will also reduce WUE, leading to impaired photosynthesis and a drop in above-ground dry matter per unit transpiration.

IN FOCUS

Chickpea Water Use Efficiency in relation to cropping system, cultivar, soil nitrogen and Rhizobial inoculation in semiarid environments

In this research, cropping system, cultivar, and inoculation all had greater impact on WUE than on the amount of water extracted by the crop from the soil. Crops grown in semiarid rainfed conditions are prone to water stress, which could be alleviated by improving cultural practices. This study determined the effect of cropping system, cultivar, soil nitrogen status and *Rhizobium* inoculation (*Rz*) on water use and Water Use Efficiency (WUE) of chickpea (*Cicer arietinum* L.) in semiarid environments.

Four varieties were grown in no-till barley, no-till wheat, and tilled-fallow systems and under various rates of N fertiliser (0, 28, 56, 84, and 112 kg N ha−1) coupled with or without *Rz*. On average, chickpea used about 10 mm of water from the top 0–15 cm soil depth. In the tilled-fallow system, chickpea extracted 20% more water in the 15–30 cm depth, 70% more in the 30–60 cm depth, and 156% more in the 60–120 cm depth than when it was grown in the no-till systems. Cropping system, cultivar, and inoculation all had greater impact on WUE than on the amount of water extracted by the crop from the soil. The improvement of cultural practices to promote general plant health along with the development of cultivars with improved crop yields will be keys for improving WUE of chickpea in semiarid environments. e 30–60 cm depth, and 156% more in the 60–120 cm depth than when it was grown in the no-till systems.

WUE increased from 4.7 to 6.8 kg ha−1 mm−1 as N fertiliser rate was increased from 0 to 112 kg N ha−1 when chickpea was grown in the no-till barley or wheat systems, but chickpea grown in the tilled-fallow system did not respond to changes in the fertiliser N rates averaging WUE of 6.5 kg ha−1 mm−1. In the absence of N fertiliser, the application of *Rz* increased WUE by 33% for chickpea grown in the no-till barley system, 30% in the no-till wheat system, and 9% in the tilled-fallow system. Chickpea inoculated with *Rhizobium* achieved a WUE value similar to the crop fertilised at 84 kg N ha−1. Without the use of *Rz*, chickpea increased WUE in a linear fashion with increasing fertiliser N rates from 0 to 84 kg N ha−1.

The improvement of cultural practices to promote general plant health along with the development of cultivars with improved crop yields will be keys for improving WUE of chickpea in semiarid environments. [58]

Crop biomass and grain yield depend on photosynthesis. Photosynthesis involves the uptake of carbon dioxide (CO_2) through stomata, which are pore-like, specialised cells

57 Oweis, T., Hachum, A., & Pala, M. (2004). Water Use Efficiency of winter-sown chickpea under supplemental irrigation in a Mediterranean environment. *Agricultural water management*, *66*(2), 163-179.

58 Gan, Y. T., Warkentin, T. D., Bing, D. J., Stevenson, F. C., & McDonald, C. L. (2010). Chickpea Water Use Efficiency in relation to cropping system, cultivar, soil nitrogen and Rhizobial inoculation in semiarid environments. *Agricultural water management*, *97*(9), 1375-1381.

in the surface of leaves. However, open stomata required for CO_2 uptake are an open gate for water loss. Therefore, there is a tight trade-off between uptake of CO_2 and water loss, and this explains the close link between crop production and water use. [59]

Managing to optimise Water Use Efficiency

Measures to improve WUE should aim to reduce soil evaporation during winter both directly, by increasing soil cover, for example, by mulches or earlier-developing canopies, and indirectly by increasing infiltration. In spring, measures to increase WUE should aim at reducing transpiration by minimising canopy development to what is required by the crop to maximize harvest index. [60]

While farmers have no control over rainfall, by using different management practices they can affect how much of the rainfall is used by the crop and how efficiently it is used.

Fallowing captures out-of-season rainfall and can increase the amount of water available for crop growth. However, the proportion of rainfall retained by fallowing (also referred to as fallow efficiency) can be small, typically of the order of 20% but frequently less.

Retaining stubbles on the fallow and controlling summer weeds may help to reduce water loss from the fallow and improve fallow efficiency, although the value of stubble retention appears to vary with soil texture and rainfall. On sandy soils, there may be little benefit from stubble retention on water capture over summer and in some cases standing stubble may enhance evaporative losses. In contrast, on clay soils in southern Australia, fallow efficiencies up to 40% have been measured with retained stubbles.

Crop choice: There are intrinsic differences in the WUE of crops, with wheat more water use efficient than grain legumes, or canola whether considered in terms of total biomass production or grain yield. Chickpeas in the Australian Mediterranean–type environment have been estimated to have a WUE based on total biomass of 16.0 (11.1–18.3) kg/ha.mm and based on grain yield of 6.2 (4.7–8.9) kg/ha.mm. [61]

Arguably, time of sowing is the most important management practice determining WUE and yield. Many studies in a range of crops have shown that 'late' sowing will reduce yields, although for short-season varieties, sowing very early may have little benefit or may reduce yields. Time of sowing generally has only a small effect on total crop water use but can have a marked effect on WUE, and the highest water use efficiencies are consistently achieved when the crop is sown at the optimum time. Late sowing reduces WUE for a number of reasons: sowing into colder soil delays crop establishment and early vigour, which increases the proportion of crop evapotranspiration lost as soil evaporation; there is a higher likelihood of heat stress around flowering and during grain growth; and there are reductions in biomass per unit water use associated with increasing vapour pressure deficit.

Recommendations for increasing water use in crop have been made based on trials exploring water use of chickpeas in Southern Australia (Table 8). Measures to improve WUE should aim to reduce soil evaporation during winter both directly, by increasing soil cover, for example, by mulches or earlier-developing canopies, and indirectly by increasing infiltration. In spring, measures to improve WUE should aim at reducing transpiration by minimising canopy development to what is required by the crop to maximise harvest index.

VIDEOS

14. GRDCTV10: Grazing stubbles and Water Use Efficiency.

59 V Sandras, G McDonald. GRDC. (2012). Water Use Efficiency of grain crops in Australia: principles, benchmarks and management.

60 Siddique, K. H. M., & Sedgley, R. H. (1987). Canopy development modifies the water economy of chickpea (*Cicer arietinum* L.) in south-western Australia. *Crop and Pasture Science*, *37*(6), 599-610.

61 Siddique, K. H. M., Regan, K. L., Tennant, D., & Thomson, B. D. (2001). Water use and Water Use Efficiency of cool season grain legumes in low rainfall Mediterranean-type environments. *European Journal of Agronomy*, *15*(4), 267-280.

Table 8: *Effect of planting date on total water use (E,) and WUE of chickpea.* [62]

Planting date	Total water use (E_t mm)	WUE (kg dry matter ha^{-1} mm^{-1})	GWUF. (kg seed ha^{-1} mm^{-1})	Planting date	Total water use (E_1 mm)	WUE (kg dry matter ha^{-1} mm^{-1})	GWUE (kg seed ha^{-1} mm^{-1})
(a) 1982				**(b) 1983**			
11 May (D1)	213.2	23.2	6.81	17 May (D1)	191.8	35.2	6.52
26 May (D2)	214.1	19.5	5.94	31 May (D2)	182.8	29.2	6.18
17 June (D3)	229.6	18.8	5.55	14 June (D3)	182.1	26.6	6.15
30 June (D4)	227.2	16.9	4.86	30 June (D4)	188.3	21.1	5.88
				20 July (D5)	182.1	17.7	5.15
Mean	221.0	19.5	5.79	Mean	185.4	26.2	5.98
l.s.d. (P<0.5)	24.8	2.5	1.09	l.s.d. (P<0.5)	17.8	5.8	1.04
C.V. (%)	7.0	10.2	11.7	C.V. (%)	6.2	12.8	11.3

Source: Siddique *et al*

MORE INFORMATION

Water Use Efficiency of grain crops in Australia: principles, benchmarks and management

VIDEOS

15. Over The Fence South: Weighing up water efficiency versus wide rows

The spatial arrangement of plants in crops, the result of the chosen row spacing and sowing rate, affects crop water use in two main ways: firstly it affects the rate of early growth and the degree and timing of canopy closure, and thus the proportion of crop water use lost as soil evaporation; and secondly it influences the partitioning of water use between the pre-anthesis and post anthesis periods. The amount and distribution of rainfall will largely influence the optimum sowing rate.

Nevertheless, grain yields (and WUE) are quite stable over a wide range of sowing rates (Table 8), which affords a degree of flexibility when deciding on the most appropriate sowing rate. Increased sowing rates increase early crop growth rate and potentially reduce evaporation from the soil surface, thus 'saving' water for use later in the season. On the other hand, high sowing rates will lead to vigorous early crop growth and water use, which may cause early depletion of soil moisture if rainfall is low. In general, using low plant densities in low-rainfall regions, or in regions where crops depend on soil moisture reserves at sowing, helps to partition water use between the pre-flowering and post-flowering periods more effectively.

Row spacing may have relatively little effect on WUE. The potential gains in WUE from altering row spacing will depend on how it affects the proportion of moisture lost from bare soil evaporation and how it influences the pattern of water use during the growing season. Increased row spacing can lead to increased exposure of the soil surface and raised soil evaporation, but where the maximum leaf area of the crop is small, or where the soil surface is not moist for long periods of time, altering row width has little effect on bare soil evaporation. Present evidence suggests that using wide rows in non-cereal crops may have limited benefit to the efficient use of seasonal rainfall or may cause reductions in efficiency. [63]

1.8.5 Nitrogen use efficiency

Nitrogen Use Efficiency (NUE) is the product of absorption efficiency (amount of absorbed N/quantity of available N) and the utilisation efficiency (yield/absorbed N). For a large number of crops, there is a genetic variability for both N absorption efficiency and for N utilisation efficiency. For abiotic stress improvement in crops, NUE has become the second priority after drought both in the private and in the public sector.

One of the main challenges in the future will be to develop reliable decision support systems with the help of sensors and biological diagnostic tools in precision

62 Siddique, K. H. M., & Sedgley, R. H. (1987). Canopy development modifies the water economy of chickpea (*Cicer arietinum* L.) in south-western Australia. *Crop and Pasture Science*, *37*(6), 599-610.

63 V Sandras, G McDonald. GRDC. (2012). Water Use Efficiency of grain crops in Australia: principles, benchmarks and management.

agriculture, in order to optimise the application of N under organic or conventional conditions in a more sustainable manner. [64]

Breeding for more efficient symbioses with Rhizobia and arbuscular mycorrhizal (AM) fungi can be an alternative for increasing plant productivity, while using the same amount of synthetic N fertiliser. Conservation tillage using no till and continuous cover cropping cultures are also known to increase significantly the potentiality and diversity of plant colonisation by AM fungi in comparison to conventional tillage. Thus, these new alternative farming techniques can increase NUE for a number of crops through the beneficial action of AM.

Under both humid conditions and dry weather, chickpea has been found to maintain a constant partial factor N use efficiency (PFNUE: grain yield per unit fertiliser N) and a consistently high N utilisation efficiency (NUtE: grain yield per unit N in the above-ground dry matter) for grain production. [65]

Whatever the mode of N fertilisation, an increased knowledge of the mechanisms controlling plant N economy is essential for improving NUE and for reducing excessive input of fertilisers, while maintaining an acceptable yield and sufficient profit margin for farmers.

There are now sufficient data from Australian and overseas studies to be able to categorise the pulses either as strong N_2 fixers (lupin and fababean), medium N_2 fixers (chickpea, field pea, mung bean and lentil) or weak N_2 fixers (navy bean).

MORE INFORMATION

Water Use Efficiency of grain crops in Australia: principles, benchmarks and management

Nitrogen Fixation Benefits of Pulse Crops

https://grdc.com.au/resources-and-publications/grdc-update-papers/tab-content/grdc-update-papers/2009/09/nitrogen-fixation-benefits-of-pulse-crops

1.9 Disease status of paddock

Three pre-planting practices are paramount for managing chickpea diseases: stubble management, controlling volunteers and weeds, and paddock selection.

Floods and surface water flows can distribute inoculum of *Phoma rabiei* (formerly *Ascochyta rabiei*, causing Ascochyta blight) and *Botrytis cinerea* (causing Botrytis grey mould) as well as *Sclerotinia*, *Phytophthora root rot* and root-lesion nematodes across large areas. Some diseases such as Ascochyta blight are considered 'community diseases', so what happens in a neighbouring paddock or even several kilometres away can affect crops. [66]

Ascochyta blight in chickpeas is now manageable, but can still loom as the biggest potential issue in Australian chickpea production unless it is managed by a combination of variety choice, strategic use of fungicides and crop hygiene (seed source, rotation, proximity of chickpea stubbles).

Chickpea crops in southern Australia and isolated parts of northern Australia are being hit by a more virulent strain of the damaging ascochyta blight.

Ascochyta resistant varieties are not immune to Ascochyta blight, particularly at pod fill, but do make it easier to control with reduced risk, inputs and expense. Lower rainfall areas must be considered as being at least medium risk for ascochyta, but could be high risk on an individual paddock basis. Know the Ascochyta blight disease rating of the variety grown; assess the individual paddock risk and manage the crop appropriately. Be aware of the specific management needs for the variety chosen through its variety management package (VMP).

Avoid sowing chickpeas into paddocks that have a recent and prolonged history of predominantly legume (e.g. medic, lentil, and field pea) or broadleaf crops (e.g. canola). Phoma, fusarium, pythium, or sclerotinia may be present.

64 Hirel, B., Tétu, T., Lea, P. J., & Dubois, F. (2011). Improving nitrogen use efficiency in crops for sustainable agriculture. *Sustainability*, *3*(9), 1452-1485.

65 Neugschwandtner, R. W., Wagentristl, H., & Kaul, H. P. (2015). Nitrogen yield and nitrogen use of chickpea compared to pea, barley and oat in Central Europe. *International Journal of Plant Production*, *9*(2), 291-304.

66 GRDC (2011) What to consider before planting chickpeas. GRDC Media Centre 6 June 2011, http://grdc.com.au/Media-Centre/Media-News/North/2011/06/What-to-consider-before-planting-chickpeas

Seed treatment should be considered mandatory for protection, especially with Kabuli types. Resistance to PRR may perhaps provide slightly better tolerance to waterlogging or common root rots. [67]

1.9.1 Cropping history effects

Paddocks closer than 1 km to last year's chickpea stubble should be considered as a higher risk from Ascochyta blight infection and need to be managed as such. Varieties with higher Ascochyta resistance such as should be considered; otherwise, there is the need for programmed Ascochyta spraying through the season. Where possible place as great a distance as practical between this year and last year's chickpea paddocks and be mindful of common wind direction. [68]

For more information, see Section 9: Diseases.

1.10 Nematode status of paddock

Cereal Cyst Nematode (CCN, *Heterodera avenae*) is a pest of graminaceous crops worldwide. This nematode is a significant problem across south-eastern Australia. CCN becomes more problematic in areas where intensive cereal cropping occurs. Cereal cyst nematode will only infect, feed and develop on cereals and other grasses. Non-cereal crops, including chickpeas, will not host the nematode, so are useful in rotations to limit damage caused to cereals. [69]

Root Lesion Nematodes (RLN) are widespread in cropping soils. Although mainly considered an issue in wheat crops, RLN also infects chickpeas, with yield losses of 20–30% previously recorded in intolerant varieties. Chickpeas are susceptible to RLN, which means that this nematode colonises the root systems and builds up numbers in the soil. However, chickpea varieties can vary in their levels of resistance to RLN; this is related to the extent to which they build up RLN populations in the soil, which then dictates the effect on subsequent crops in the rotation. Varieties that are more susceptible allow greater multiplication of Pt in their root systems over a season. The higher the resulting RLN population left in the soil following chickpeas, the greater is the potential for a negative impact on the yield of subsequent crops.

1.10.1 Effects of cropping history on nematode status

Nematode numbers build up steadily under susceptible crops and cause decreasing yields over several years. Yield losses >50% can occur in some wheat varieties due to RLNs, and up to 20% in some chickpea varieties. The amount of damage caused will depend on:

- the numbers of nematodes in the soil at sowing
- the tolerance of the variety of the crop being grown
- the environmental conditions.

Generally, a population density of 2 RLN species/g soil anywhere in the soil profile has the potential to reduce the grain yield of intolerant varieties. A tolerant crop yields well when high populations of nematodes are present (the opposite is intolerance). A resistant crop does not allow nematodes to reproduce and increase in number (the opposite is susceptibility).

Growing resistant crops is the main tool for managing nematodes. In the case of crops such as chickpea, choose the most tolerant variety available and rotate with resistant crops to keep nematode numbers at low levels. Information on the responses of crop varieties to nematodes is regularly updated in grower and planting

67 Pulse Australia. Southern Pulse Bulletin PA 2010 #05 – Chickpea checklist for southern growers. http://www.pulseaus.com.au/storage/app/media/crops/2010_SPB-Chickpea-checklist-south.pdf

68 Pulse Australia. Southern Pulse Bulletin PA 2010 #05 – Chickpea checklist for southern growers. http://www.pulseaus.com.au/storage/app/media/crops/2010_SPB-Chickpea-checklist-south.pdf

69 Soilquality.org. (2016). Cereal cyst nematode. http://www.soilquality.org.au/factsheets/cereal-cyst-nematode

guides. Note that crops and varieties have different levels of tolerance and resistance to different nematode species. [70]

For more information, see Section 8: Nematodes.

1.11 Testing soil for disease and nematodes

Diseases and nematodes that occur underground can be difficult to detect and diagnose but they must be identified correctly to enable appropriate control measures to be implemented. It is important to have paddocks diagnosed for plant parasitic nematodes and disease so that optimal management strategies can be implemented. Testing your farm will tell you if nematodes or disease are present in your paddocks and at what density as well as which species are present.

It is important to know which species are present because some crop-management options are species-specific. If a particular species is present in high numbers, immediate decisions must be made to avoid losses in the next crop to be grown. With low numbers, it is important to take decisions to safeguard future crops. Learning that a paddock is free of these nematodes is valuable information because steps may be taken to avoid its future contamination.

Testing of soil samples taken either before a crop is sown or while the crop is in the ground provides valuable information. There is a great deal of spatial variation in nematode populations within paddocks. It is critical to follow sampling guidelines to ensure accurate results. [71]

Root diseases that are most likely to be encountered under intensive cereal cropping systems are rhizoctonia bare patch, root lesion nematode, take-all, fusarium crown rot, cereal cyst nematode, common root rot, and pythium root rot.

Diseases caused by pathogenic micro-organisms can significantly reduce the yield of cereals. The most prevalent root diseases of cereals in southern Australia are rhizoctonia bare patch, root lesion nematode, fusarium crown rot, and take-all. Less widespread are cereal cyst nematode, common root rot, and pythium root rot.

It is crucial that these diseases are correctly diagnosed to enable the right control measures to be employed for the benefit of crop yields.

1.11.1 Soil and plant testing services for diagnosing root diseases

Firstly, look at the distribution of symptomatic plants throughout the whole crop.

To determine whether a fungal or nematode root disease is affecting a cereal crop, look for patchy areas of poor crop development associated with localised disease build up. Some root disease such as fusarium crown rot may be more evenly scattered or distributed throughout the crop.

Next, carefully dig up samples of apparently diseased as well as healthy plants.

Thoroughly wash the soil from the roots and examine them for indicative symptoms, which vary to some extent depending on the disease. Unthrifty plants may have smaller root mass, fewer root branches, root browning, root clumping, or damaged root tips (spear tips) compared to thrifty or well grown plants nearby.

The MyCrop app may assist you with diagnosis.

MORE INFORMATION

Some recent research papers are available online:

Rhizoctonia solani AG8: New breakthroughs in control and management, 2015 Crop Updates paper

Liquid banding of fungicide increases yields of cereals in paddocks with rhizoctonia bare-patch, 2014 Crop Updates paper

Rhizoctonia bare patch in cereals - Integrated disease management options, 2013 Crop Updates paper

Root lesion nematode has a picnic in 2013, 2014 Crop Updates paper

Pratylenchus teres - WA's home grown Root Lesion Nematode, 2013 Crop Updates paper

For further information refer to Root lesion and burrowing nematodes: diagnosis and management.

70 K Owen, T Clewett, J Thompson (2013) Summer crop decisions and root-lesion nematodes. GRDC Update Papers 16 July 2013, http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/07/Summer-crop-decisions-and-root-lesion-nematodes

71 S Simpfendorfer, M Gardner, G McMullen (2013) Desi chickpea varieties differ in their resistance to the root lesion nematode Pratylenchus thornei—Come-by-Chance 2010. Northern Grains Region Trial Results, autumn 2013. pp. 114–116. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0004/468328/Northern-grains-region-trial-results-autumn-2013.pdf

Confirmation of diagnosis

Suspected root disease or nematode problems in-crop can be confirmed by laboratory analysis of soil and/or roots. For patch diseases, sample from the edge of the patch rather than the centre.

Pre-season assessment:

The risk of root diseases being present in a paddock at a yield limiting level next season can be determined by paddock history and paddock monitoring in spring or soil tests. A review of paddock history will identify the diseases likely to be present in each paddock. The level of disease likely to develop can be determined by digging up plants in spring from areas of poor growth and examining the roots for symptoms.

An informed decision can be made about the future use of each paddock based on the presence or absence of a disease and the conduciveness of the current season and crop to further develop that disease.

Pre-season soil tests can be used where the paddock history is not adequate for planning future use. Soil tests are conducted on representative soil samples. PreDicta-B™ uses DNA assessment to determine the root diseases or nematode species present and the likely risk of crop damage. Test kits are available through accredited agronomists and resellers. [72]

PreDicta B (B = broadacre) is a DNA-based soil testing service to identify which soil-borne pathogens pose a significant risk to broadacre crops prior to seeding.

It has been developed for cropping regions in Australia and includes tests for:

- cereal cyst nematode (CCN)
- take-all (*Gaeumannomyces graminis* var. *tritici* (Ggt) and *G. graminis* var. *avenae* (Gga))
- rhizoctonia barepatch (*Rhizoctonia solani* AG8)
- crown rot (*Fusarium pseudograminearum*) *Note that oats are not very susceptible to crown rot – but does host it.
- root lesion nematode (*Pratylenchus neglectus* and *P. thornei*)
- stem nematode (*Ditylenchus dipsaci*).

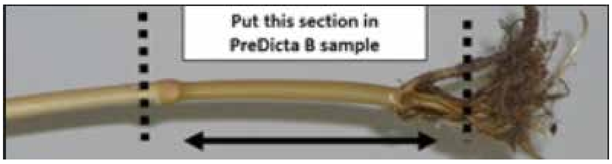


Photo 12: *Correct sampling strategy.*

Source: GRDC.

MORE INFORMATION

PIR SA Predicta B website

PreDicta B samples are processed weekly from February to mid-May (prior to crops being sown) to assist with planning the cropping program. PreDicta B is not intended for in-crop diagnosis. That is best achieved by sending samples of affected plants to your local plant pathology laboratory.

1.12 Insect status of paddock

Deciding the best way to sample for a particular pest depends on where in the crop the pest feeds and shelters, and the effects of weather on its behaviour. The stage of crop development and the insect being monitored will determine which sampling method is most suitable. For example, pests in seedling crops generally cannot be collected by sweeping because the crop is too short.

Pest outbreaks occur often in response to natural conditions, but sometimes in response to management practices. Minimum tillage and stubble retention have

72 DAFWA (2016). Root disease under intensive cereal production systems. https://www.agric.wa.gov.au/barley/root-disease-under-intensive-cereal-production-systems

resulted in greater diversity of invertebrate species seen in crops. Cultural control methods such as burning, rolling, or cultivating stubbles are sometimes needed to complement chemical and biological controls. [73]

Soil-dwelling insect pests can seriously reduce plant establishment and populations, and subsequent yield potential.

Soil insects include:

- cockroaches
- crickets
- earwigs
- black scarab beetles
- cutworms
- false wireworm
- true wireworm

Different soil insects occur under different cultivation systems and farm management can directly influence the type and number of these pests:

- Weedy fallows and volunteer crops encourage soil insect build-up.
- Insect numbers decline during a clean long fallow due to lack of food.
- High levels of stubble on the soil surface can promote some soil insects due to a food source, but this can also mean that pests continue feeding on the stubble instead of germinating crops.
- No-tillage encourages beneficial predatory insects and earthworms.
- Incorporating stubble promotes black field earwig populations.
- False wireworms are found under all intensities of cultivation but numbers decline if stubble levels are very low.

Soil insect control measures are normally applied at sowing. Since different insects require different control measures, the species of soil insects must be identified before planting. Soil insects are often difficult to detect as they hide under trash or in the soil. Immature insects such as false wireworm larvae are usually found at the moist/dry soil interface.

1.12.1 Insect sampling of soil

Sampling methods should be applied in a consistent manner between paddocks and sampling occasions. Any differences can then be confidently attributed to changes in the insect populations, and not different sampling techniques. [74]

Soil sampling by spade

1. Take a number of spade samples from random locations across the field.
2. Check that all spade samples are deep enough to take in the moist soil layer (this is essential).
3. Hand-sort samples to determine type and number of soil insects.

Germinating-seed bait technique

Immediately following planting rain:

1. Soak insecticide-free crop seed in water for at least two hours to initiate germination.
2. Bury a dessertspoon of the seed under 1 cm of soil at each corner of a 5 m by 5 m square at five widely spaced sites per 100 ha.
3. Mark the position of the seed baits, as large populations of soil insects can destroy the baits.
4. One day after seedling emergence, dig up the plants and count the insects.

73 https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/07/Emerging-insect-pests

74 http://ipmguidelinesforgrains.com.au/insectopedia/introduction/sampling.htm

Trials have shown no difference in the type of seed used for attracting soil-dwelling insects. However, use of the type of seed to be sown as a crop is likely to indicate the species of pests that could damage that crop. The major disadvantage of the germinating-grain bait method is the delay between the seed placement and assessment. [75]

The South Australian Research and Development Institute (SARDI) Entomology Unit provides an insect identification and advisory service. The Unit identifies insects to the highest taxonomic level for species where this is possible and can also give farmers biological information and guidelines for control. [76]

PestNotes

PestNotes are designed specifically for growers, agronomists and farm advisers. They bring together the best available information and images on more than 50 invertebrate pests of the southern cropping region. These information sheets have been developed through a collaboration between cesar and the South Australian Research and Development Institute.

Insect ID: The Ute Guide

The Insect ID Ute Guide is a comprehensive reference guide for insect pests commonly affecting broadacre crops and growers across Australia, and includes the beneficial insects that may help to control them. Photos have been provided for multiple life-cycle stages, and each insect is described in detail, with information on the crops they attack, how they can be monitored, and other pests that they may be confused with. Use of this app should result in better management of pests, increased farm profitability, and improved chemical usage. [77]

App Features:

- Region selection
- Predictive search by common and scientific names
- Compare photos of insects side by side with insects in the app
- Identify beneficial predators and parasites of insect pests
- Opt to download content updates in-app to ensure you're aware of the latest pests affecting crops for each region
- Ensure awareness of international bio-security pests

Insect ID, The Ute Guide is available on Android and iPhone.

It is important to consider paddock history when planning for pest management. Resident pests can be easier to predict by using paddock history and agronomic and weather data to determine the likely presence (and numbers) of certain pests

75 DAFF (2011) How to recognise and monitor soil insects. Queensland Department of Agriculture, Fisheries and Forestry, https://www.daf.qld.gov.au/plants/field-crops-and-pastures/broadacre-field-crops/integrated-pest-management/help-pages/recognising-and-monitoring-soil-insects

76 http://pir.sa.gov.au/research/research_specialties/sustainable_systems/entomology/insect_diagnostic_service

77 https://grdc.com.au/Resources/Apps

MORE INFORMATION

Pest Genie Australia

APVMA

within a paddock. This will point towards the likely pest issues and allow growers to implement preventative options. [78]

Reduced tillage and increased stubble retention have changed the cropping landscape with respect to soil moisture retention, groundcover, and soil biology and this has also affected the abundance and types of invertebrate species being seen in crops. These systems increase invertebrate biodiversity but also create more favourable conditions for many pests such as slugs, earwigs, weevils, beetles, and many caterpillars. In turn they have also influenced beneficial species such as carabid and lady beetles, hoverflies, and parasitic wasps. [79]

See Section 7: Insect control for more information.

78 https://grdc.com.au/Media-Centre/Ground-Cover/Ground-Cover-Issue-117-July-August-2015/Growers-chase-pest-control-answers

79 https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/07/Emerging-insect-pests

Pre-planting

MORE INFORMATION

Checklist for Chickpea growers in the Southern region.

Key messages

- In variety choice, consider yield and adaptation to the area, disease resistance, grain quality, marketability, and proximity to receival point.
- Be aware of the specific management needs for the variety chosen through its Variety Management Package (VMP). [1]
- Kabuli chickpea provides a very profitable cropping option to southern Australian grain growers when produced under the right conditions and management.
- Test for seed quality in terms of germination and vigour prior to sowing. Only sow the highest quality seed.
- If using grower retained seed, ensure that it has been stored correctly; i.e. at the right moisture and temperature and for no longer than 12 months.

FAQ

2.1.1 Choosing a variety

Choosing a variety that has been bred for, and proven in, the southern grains region is the first step in successful chickpea production (Photo 1). Understanding varietal ratings with respect to diseases and their control is a key part of risk management.

Photo 1: *Chickpea trials test for varietal differences.*

Source: W.Hawthorne.

The availability of varieties resistant to Ascochyta blight now provides growers with low disease-risk options for growing chickpeas in southern Australia. Ascochyta blight of chickpeas has been a widespread and devastating disease in all Australian grain regions, and unless resistant varieties are used, it can be a major limitation when growing this crop.

Chickpea crops in southern Australia and isolated parts of northern Australia are being hit by a more virulent strain of the damaging ascochyta blight. Pulse pathologists in Victoria and South Australia have noted a marked decline in the

1 Pulse Australia. Southern Pulse Bulletin PA 2010 #05 – Chickpea checklist for southern growers. http://www.pulseaus.com.au/storage/app/media/crops/2010_SPB-Chickpea-checklist-south.pdf

resistance of several varieties of chickpeas, with varieties previously rated as moderately resistant performing like susceptible lines. [2]

Growers should check for revised AB resistance ratings and be prepared to monitor and act on disease control if AB disease levels are higher than expected during the season.

During 2015 at Curyo (southern Mallee), in early August a significant outbreak of ascochyta blight was observed in a kabuli chickpea trial. Symptom assessment indicated that this isolate of ascochyta was different from those observed previously in Victorian trials, having virulence on resistant lines such as Genesis090™ and PBA Slasher⍉. From the results in this trial, there appears to be some differences in resistance to this isolate with CICA1454 showing fewer symptoms and PBA Striker⍉ being significantly affected. [3]

Some varieties with Ascochyta blight resistance that are available to growers may have other agronomic, disease or marketability limitations and will not suit all areas or situations.

When choosing varieties to grow, it is essential to consider their susceptibility to Ascochyta blight and Phytophthora root rot, along with yield potential, price potential, marketing opportunities, flowering cold tolerance, maturity timing, lodging resistance, and other agronomic features relevant to your growing region.

Disease management is a major factor in chickpea cropping and with proactive management, foliar diseases can be managed to optimise potential yield.

When comparing yields between varieties, growers need to be aware that where Ascochyta blight pressure is high, varieties with moderate resistance, or less, are more likely to suffer greater yield losses than the resistant lines, even with regular applications of foliar fungicides.

For details on disease ratings for chickpea varieties, see Section 9 - Diseases, Table 3.

Area of adaptation

Chickpea varieties are bred for and selected in a range of environments. Hence, individual varieties have specific adaptations to help maximise yield and reliability under particular conditions. These conditions include rainfall, geography, temperatures, disease pressure, and soil type.

The national chickpea area has been categorised by Pulse Breeding Australia (PBA) into five regions based on rainfall and geographic location (Figure 1). The Southern Australia growing region includes:

- Region 4; medium/high rainfall mediterranean/temperate
- Region 5; Low/medium rainfall mediterranean/temperate

2 Heard G. (2016). Ascochyta pressure on chickpeas http://www.stockandland.com.au/story/4085979/ascochyta-pressure-on-chickpeas/

3 Brand J, Rodda M, Rosewarne G, Delahunty A, Kimber R, Davidson J, Hobson K. (2016). GRDC Update Papers: Your future pulse - pulse breeding and agronomy update https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/07/Your-future-pulse-pulse-breeding-and-agronomy-update

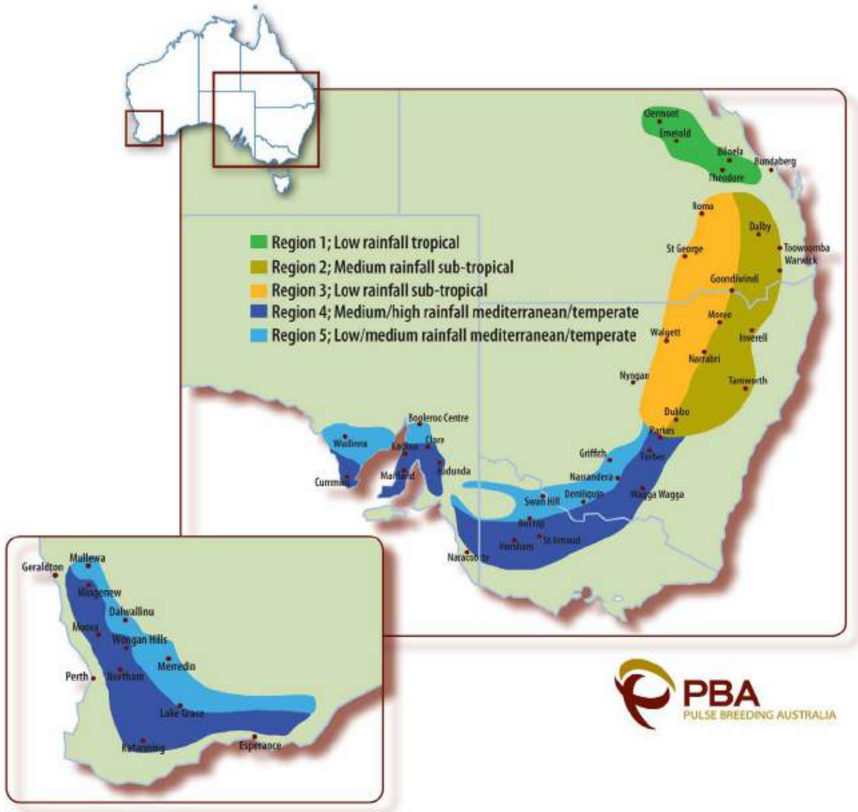


Figure 1: *Area of adaptation of chickpeas across Australian grain growing regions.*

Source: GRDC.

These regions cross state borders and are target zones for national breeding programs and variety evaluation. Breeding trials and National Variety Trial (NVT) results help indicate specific adaptation even within a region. The area of adaptation is specified for each variety so that potential users are aware of their best fit. [4]

New opportunities for pulses in the Mallee

A key focus of the breeding programs has been improving yield (profit) stability across a range of soil types and environments. In the 'tough and dry seasons' in each of the pulse crops, through improved varieties and agronomy growers aim to achieve the economic breakeven level which generally ranges between about 0.3 and 0.8 t/ha, dependent on commodity price.

1. **Disease resistance**: Improving disease resistance for the Mallee is about minimising the potential for increased input costs associated with management. It also enables optimisation of other practices, such as earlier sowing to maximise yield potential.
2. **Improved vigour and biomass**: In the Mallee, early vigour and biomass development in cold winter conditions is important to ensure growers can maximise yield potential. It can also aid with weed management through improved competition. However, higher vigour and biomass can also create increased disease pressure, due to earlier canopy closure, hence the importance of disease resistance and potentially agronomic practices, such as wider row spacings.
3. **Changed flowering and maturity characteristics**: The primary aims here have been to minimise risks from frosts and major heat events occurring during the flowering and podding phase and then mature early enough to maximise yield and allow the potential of crop topping to prevent weed seed set. This has led

4 G Cumming (2014) Chickpea varieties selecting horses for courses. GRDC Update Papers 5 March 2014, http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Chickpea-varieties-selecting-horses-for-courses

to the development of varieties that are generally classified as flowering in the early to mid-range with a similar maturity range. In Victorian Mallee environments, early vigour and biomass development has been essential to maximise any gain achieved through early flowering.

4. **Boron and salt tolerance**: This has been a long term aim of the breeding programs, to help alleviate soil variability that can occur throughout Mallee landscapes.
5. **Improved pod height and harvestability**: There has been significant improvements in both pod height and harvestability for all pulse crops and it continues to be an important goal in PBA breeding programs. On lighter Mallee soils it is also acknowledged that it is important to ensure there is sufficient stubble remaining in the paddock to prevent erosion events.

The value of agronomic management

While new traits are important, it is essential to remind ourselves of the value of optimum agronomic management of pulses. While stubble retention aids harvestability, yields can be more than doubled, in dry season in a retained stubble system versus a removed stubble situation.

Evaluation of yield potential

When choosing a variety many factors must be considered, including disease susceptibility, paddock suitability, seed availability, seed size and sowing rate (with reference to sowing machinery), seed cost, harvesting ease, and marketing options.

The most accurate predictor of a variety's performance is a stable yield in many locations over several years. Yield results from Pulse Breeding Australia (PBA) and National Variety Trials (NVT) are available from the NVT website, as well as from the specific Pulse Variety Management Package (VMP) brochure. [5]

Long-term yields can be represented in several different ways but are typically displayed either as site-specific, averaged over multiple years, or for each year averaged over multiple sites for a region.

In association with Pulse Australia and its commercial seed partners, PBA launches its new varieties at targeted pulse field days during the spring field-day circuit. This gives growers and advisors the opportunity to view and assess the varieties in their growing regions prior to their availability.

A Variety Management Package (VMP) is released with each new PBA variety. The brochures provide information about appropriate agronomic and disease management and disease ratings for each variety.

The information in the brochures is compiled from agronomic and disease management projects funded by the Grains Research and Development Corporation (GRDC) in conjunction with the PBA partner agencies, combined with yield data from variety trials conducted by both PBA and NVT. [6]

MORE INFORMATION

http://www.pulseaus.com.au/growing-pulses/bmp/chickpea

5 G Cumming (2014) Chickpea varieties selecting horses for courses. GRDC Update Papers 5 March 2014, https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Chickpea-varieties-selecting-horses-for-courses

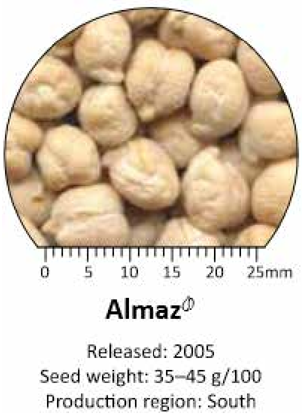
6 Pulse Breeding Australia. PBA Varieties and brochures. GRDC Major Initiatives, http://www.grdc.com.au/Research-and-Development/Major-Initiatives/PBA/PBA-Varieties-and-Brochures

2.1 Varietal performance and ratings yield

Table 1: *Varieties and varietal traits for chickpeas in the southern growing region.*

Variety	Yield ability	Quality traits	Maturity	Other varietal traits
DESI				
PBA StrikerΦ 	Produces high yields in the low to medium rainfall areas of southern Australia. Yields of PBA StrikerΦ are substantially higher (7–13%) than PBA SlasherΦ and Genesis 836.	Suitable for both splitting and direct consumption use by traders in India and the Middle East. Medium-sized Desi seed with excellent milling quality, larger seed size than PBA SlasherΦ and Genesis 836.	Improved early vigour compared to PBA SlasherΦ, with early flowering and early maturity. PBA StrikerΦ flowers 5–7 days earlier than PBA SlasherΦ and is earlier maturing than PBA SlasherΦ and Genesis 836.	Susceptible to Ascochyta blight, it is a semi-spreading plant type similar to PBA SlasherΦ. Plant height and lowest pod height is similar to PBA SlasherΦ but lower than Genesis 836.
PBA SlasherΦ 	Excellent adaptation to all areas of Southern Australia producing high yields.	Assessed as suitable for both splitting and direct consumption use by traders in India and the Middle East. Medium- sized Desi seed with good milling quality, with higher dehulling efficiency and dhal (splits) yield than Genesis 836.	Mid-flowering and mid-maturity. PBA SlasherΦ flowers 3–7 days earlier than Genesis 836 and is earlier maturing than Genesis 836.	Moderately susceptible to Ascochyta blight, high yield and good seed quality. Semi-spreading plant type. Plant height and lowest pod height is lower than Genesis 836. Susceptible (S) to Botrytis grey mould (BGM), similar to Genesis varieties.
NeelamΦ 	Highest yielding Resistant (R) Ascochyta blight rated Desi chickpea variety. Yields of NeelamΦ are 2–5% higher than PBA SlasherΦ.	Desi type chickpea suitable for the whole and splitting human food markets. Seed size is 17 grams/100 grams (g), marginally larger than Genesis 836 similar to PBA SlasherΦ. However its seed coat colour is lighter than PBA SlasherΦ.	Mid-flowering and mid-maturity variety similar to Genesis 836.	Moderately susceptible Ascochyta blight rated. Medium/tall plant height, taller than PBA SlasherΦ.

Variety	Yield ability	Quality traits	Maturity	Other varietal traits
Ambar⍉ 	Well adapted to most of Southern Australia,. Ambar⍉ yields are similar to PBA Slasher⍉.	Desi type chickpea suitable for the whole and splitting human food markets. Seed size is similar to Genesis 836 at 16 g/100 g. However, its seed coat colour is lighter than PBA Slasher⍉.	Earliest flowering and earliest maturing of all current Desi chickpea varieties making it particularly well-suited to shorter growing season (low rainfall) environments.	Rated as moderately susceptible to Ascochyta blight. Bushy growth habit with profuse branching it has a short/medium plant height slightly shorter than PBA Slasher⍉.
PBA Maiden⍉ 	It is broadly adapted to these regions and has shown similar yields to PBA Slasher⍉.	Largest seed size of current southern Desi chickpea varieties (28% larger than PBA Slasher⍉). Targeted for whole seed markets.	Moderate early vigour. Early to mid-flowering and maturity. Semi spreading plant type.	Susceptible to Ascochyta blight.
KABULI				
Genesis™ 079 	Highest yields in short season environments than current varieties.	Budget for grain prices at lowest end of small Kabuli range due to 6–7 mm seed.	A small seeded Kabuli (predominantly 6–7 mm) with smaller seed than Genesis™090 (predominantly 6–7 mm). Early maturity and uniform short plant height offers improved potential for agronomic weed control options under some conditions.	Susceptible to foliar Ascochyta blight. A small seeded Kabuli (predominantly 6–7 mm) with smaller seed than Genesis™090 (predominantly 6–7 mm). Susceptible to phytophthora.

Variety	Yield ability	Quality traits	Maturity	Other varietal traits
Genesis ™ 090  	Broadly adapted with high yield potential. Grain prices will be lower than large Kabuli price but usually higher than a Desi price.	Genesis 090 has predominantly 7–8 mm seed, which is smaller than Almaz⁽ᵈ⁾ (8–10 mm). Suits many farming systems including inter-row sowing into standing stubbles and wider (>30 cm) row spacings.	It is medium flowering and maturing, which makes it suitable for most growing areas.	Moderately susceptible to ascochyta blight. Small Kabuli chickpea in southern Australia. Not suitable for crop topping and its height makes it unsuitable for weed wiping.
Genesis ™ 114  	Yields are higher than Almaz⁽ᵈ⁾ and Kaniva. Lower yielding and more susceptible to Ascochyta blight than Genesis 090.	Genesis 114 is a medium-sized Kabuli type chickpea (predominantly 8–9 mm). Is suited to regions with 400–700 mm annual rainfall. Medium to tall plant height with an erect plant type which is resistant to lodging. Produces cream coloured seed, larger than Genesis 090 but slightly smaller than Almaz⁽ᵈ⁾ (predominantly 8–9 mm).	Flowering and maturity time are similar to Almaz⁽ᵈ⁾ but later than Genesis 090.	Susceptible to Ascochyta blight.
Almaz⁽ᵈ⁾  	Yield greater than current large seeded Kabuli chickpea and previous standard varieties (Kaniva).	Greater seed size than Kaniva but smaller than Nafice. Attractive beige coat similar to Kaniva.	Almaz⁽ᵈ⁾ starts flowering on average one day earlier than Kaniva with similar maturity.	Moderately susceptible to ascochyta blight. Moderately susceptible to PRR.

Variety	Yield ability	Quality traits	Maturity	Other varietal traits
Kalkee 	Yields are equal to Genesis114 and Almaz$^{\phi}$ but seed size is larger. Improved grain weight (45 g/100 seeds) compared to Genesis 090 (33 g/100 seeds).	Is a large-sized Kabuli type chickpea (predominantly 9 mm) subject to rainfall at podset. Is suited to regions with 400–700 mm annual rainfall. Medium to tall plant height with an erect plant type which is resistant to lodging. Sets pods high in canopy (increased height to lowest pod than Genesis 090). Produces cream coloured, large seed, larger than Genesis 090 and Almaz$^{\phi}$.	Flowering and maturity time are slightly later than Almaz$^{\phi}$.	Moderately susceptible to ascochyta blight.
PBA Monarch$^{\phi}$ 	Highest yielding medium-sized Kabuli chickpea in all Kabuli growing areas of Australia.	Predominantly 8–9 mm seed size (larger than Genesis 090 and similar to Almaz$^{\phi}$). Semi-spreading plant type.	Early flowering and maturity (earlier than Genesis 090 and Almaz$^{\phi}$).	Susceptible to Ascochyta blight (similar to Almaz$^{\phi}$ and Kalkee but more susceptible than Genesis 090). Susceptible (S) to Phytophthora root rot.

Sources: Pulse Australia

Table 2: *Chickpea variety agronomic guide*

Variety	Ave 100 seed wt (g)	Seed size group	Early growth	Flowering	Maturity	Height	Lodging resistance
DESI							
Ambar$^{\phi}$	16	small	poor-mod	early	early	short-mid	MS
Howzat$^{\phi}$	20	medium	poor-mod	mid	mid	mid	MS
Neelam$^{\phi}$	17	medium	mod	mid	mid	short-mid	MS
PBA Maiden$^{\phi}$	24	med-large	mod	mid	mid	short-mid	MS
PBA Slasher$^{\phi}$	18	medium	poor-mod	mid	mid	short-mid	MS
PBA Striker$^{\phi}$	22	medium	good	early	early	short-mid	MS
KABULI							
Almaz$^{\phi}$	38	medium	mod	mid-late	late	mid-tall	MR
Genesis 079	24	small	good	early	early	short	MR
Genesis 090	31	small	good	mid	mid-late	mid	MR
Genesis Kalkee	45	large	good	late	late	tall	R
PBA Monarch$^{\phi}$	40	medium	poor-mod	early	early	mid	MS

Source: Victorian Winter Sowing Guide 2016.

2.2 Planting seed quality

High quality seed is essential to ensure the best start for your crop. Grower-retained seed may be of poor quality with reduced germination and vigour, as well as potentially being infected with seed-borne pathogens. No matter whether chickpea seed is acquired commercially or grower retained on-farm, it is important that it is of the highest possible quality. Poor quality seed can result in low plant establishment due to poor germination, vigour and/or seed-borne diseases such as Ascochyta blight and Botrytis grey mould (BGM).

- All seed should be tested for quality including germination (high germination—above 80%) and vigour (AA test). Use large, graded seed.
- Use seed at low risk of Ascochyta blight infection.
- If grower retained seed is of low quality, then consider purchasing registered or certified seed from a commercial supplier and always ask for a copy of the germination report.
- Regardless of the source, evenly treat seed with a thiram-based fungicide.
- Careful attention should be paid to the harvest, storage and handling of grower retained seed intended for sowing.
- Calculate seeding rates in accordance with seed quality (germination, vigour, and seed size).

High quality seed is vital (Photo 2). Check seed labels for germination percentage and purity and ask for the germination certificate. The results of a germination test must be supplied with all seed for sale. Take the additional precaution of having the seed tested for both Ascochyta and Botrytis grey mould. Harvesting on time minimises the development of disease on seed. [7]

Photo 2: *Poor quality seed (left) all sown on the same day. Note the lack of vigour.*

Source: Pulse Australia.

Effect of poor quality seed on yield

Often, seed quality problems only emerge if the crop is not harvested under ideal moisture or seasonal finishing conditions. A sharp seasonal finish, a wet harvest, or delayed harvest can have a big impact on seed quality.

Using severely weather damaged or mechanically damaged seed will result in:

- Poor establishment and poor crop performance.

7 Pulses Australia. Chickpea Production: Southern and Western region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

- Reduced plant vigour (increased susceptibility to soil-borne pathogens during establishment and increased susceptibility to foliar pathogens).
- Patchy, uneven plant stands (increased susceptibility to weed competition, aphids and viruses).
- Uneven plant development complicating in-crop management (e.g. herbicide applications).
- Uneven and delayed crop maturity (e.g. making desiccation timing difficult and leading to a mixed grain sample).
- Lower yields from a combination of all of the above.

As a general guide, weather-damaged seed with lower than normal germination and vigour levels should only be sown under very good conditions (for rapid establishment) and at a higher seeding rate. Such seed is not recommended for deep sowing or moisture seeking. [8]

The large size or fragile nature of pulse seed, particularly albus, Kabuli chickpea and faba bean, makes them more vulnerable to mechanical damage during harvest and handling. This damage is not always visually apparent and can be reduced by operations such as slowing header drum speed and opening the concave, or by reducing auger speed and lowering the flight angle and fall of grain. A rotary header and a belt elevator are ideally suited to pulse grain and can reduce seed damage, which can result in abnormal seedlings that germinate but do not develop further.

MORE INFORMATION

Chickpea: High Quality Seed

Under ideal conditions, abnormal seedlings may emerge but lack vigour, making them vulnerable to other rigours of field establishment. Factors such as low temperature, disease, insects, seeding depth, and soil crusting and compaction are more likely to affect the establishment of weak seedlings. Those that do emerge are unlikely to survive for long, will produce little biomass and make little or no contribution to final yield. [9] Sow according to germination test for satisfactory populations.

Grower retained seed

Grower-retained sowing seed should always be harvested from the best part of the crop where weeds and diseases are absent and the crop has matured evenly. Seed should be harvested first to avoid low-moisture grain, which is more susceptible to cracking. Seed moisture of 11–13% is ideal. Weeds, other grains, or disease contamination from other pulse crops should be avoided when selecting parts of the paddock for seed harvest.

Seed quality may be adversely affected by several factors including:

- Early desiccation resulting in high levels of green immature seed and smaller seed size (affecting both germination and vigour).
- Cracking of the seed coat if the seed is exposed to several wetting and drying cycles. As the seed coat absorbs moisture it expands and then contracts as it dries. This weakens the coat increasing the risk of mechanical damage during harvesting and handling operations.
- Mechanical damage can result in reduced germination and vigour and increased susceptibility to fungal pathogens in the soil at sowing (exacerbated if establishment is delayed into cold wet soils).
- Delayed harvest due to wet weather can lead to increased (i) heliothis damage; (ii) mould infection; and (iii) risk of late Ascochyta seed infection.
- Harvesting at a moisture content >15% can lead to problems with moulds and fungal pathogens colonising on the seed coat during storage.
- Harvesting at a moisture content <10% can result in mechanical damage to the seed coat and/or seed splitting, which is compounded each time the seed is handled.

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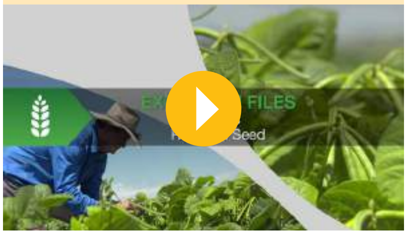
8 L Jenkins, K Moore, G Cumming. Pulse Australia. Chickpea: High Quality seed. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/high-quality-seed

9 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013, Pulse Australia Limited.

VIDEOS

1. GCTV16: Extension Files – Retained Pulse Seed.



- Poor (temporary) storage in the rush to get harvest done in wet weather can reduce viability of the seed, resulting in poor germination and emergence.
- Using chickpea seed which was shot or sprouted when harvested is not recommended. Chickpea is a relatively large seed which requires considerable moisture for germination to begin. Unlike cereal grains with higher starch content, once the chemical process has started, the seed will continue to germinate, use up its energy reserves and die due to lack of follow-up moisture whilst in storage.
- Seed-borne diseases such as Ascochyta blight and Botrytis grey mould, and/or in contamination with sclerotinia, all reduce the viability of the seed (germination and vigour). Crop establishment is reduced and any surviving infected seedlings act as an inoculum source to initiate disease infection within the new crop. [10]

NOTE: Do not use grain for seed of pulse crops harvested from a paddock that was desiccated with glyphosate. Germination, normal seedling count and vigour are affected by its use. Read the glyphosate label.

Grading

Chickpea seed for sowing should be professionally graded (preferably using a gravity grader) to provide a uniform seed lot and remove all small or split seeds, trash and weed seeds. A gravity grader is more efficient at removing lighter seeds, which are often disease infected. A higher proportion of small, shrivelled and light-weight grain can be expected in crops that have experienced high incidences of Ascochyta and or Botrytis grey mould.

Handling seed

The large size, awkward shape, and fragile nature of many pulses mean that they need careful handling to prevent seed damage. The bigger the grain, the easier it is to damage. Seed grain, in particular, should be handled carefully to ensure good germination.

- Plan ahead so that handling can be kept to a minimum to reduce damage between harvest and seeding.
- Augers with screen flighting can damage pulses, especially larger seeded types such as broad beans. This problem can be partly overcome by slowing down the auger.
- Tubulators or belt elevators are excellent for handling pulses, as little or no damage occurs. Cup elevators are less expensive than tubulators and cause less damage than augers. They have the advantage of being able to work at a steeper angle than tubulators. However, cup elevators generally have lower capacities.
- Augering from the header should be treated with as much care as later during handling and storage because it has the same potential for grain damage.
- Combine loaders that throw or sling rather than carry the grain can cause severe damage to germination.

2.2.1 Testing for seed quality

The only way to accurately know the seed's germination rate, vigour, and disease level is to have it tested. Seed-borne diseases can lower germination levels, and testing for presence in seed can be conducted by specialist laboratories for a number of diseases such as Ascochyta blight in chickpeas. Seed with poor germination potential or high levels of seed-borne disease should not be sown. Cheaper costs of this seed will be offset by higher sowing rates needed to make up for the lower germination and there is potential to introduce further disease on to the property.

10 L Jenkins, K Moore, G Cumming. Pulse Australia. Chickpea: High Quality seed. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/high-quality-seed

2.2.2 Seed size

Seed size can affect the seedling vigour in grain legumes where large seeds produce more vigorous plants and may affect yield. [11]

Seed size can be quickly calculated at home by counting out 200 seeds and weighing—multiply the weight (grams) by five to get a 1000-seed weight. Or divide 200,000 by the weight (grams) to get number of seeds per kilogram (depending on your preferred method of calculating seeding rates).

It is recommended that both the germination test and seed size test be done on several lots of seed (i.e. at least twice) to get a more accurate assessment of the sample.

2.2.3 Seed germination and vigour

Chickpeas often have poor seedling vigour which may reduce their competitiveness with weeds and pests. Low seedling vigour can also cause low WUE because of the long time to full canopy closure. Efforts that lead to improved seedling vigour among grain legumes may therefore be useful to improve yield and WUE. It has been suggested that there are inherent genetic differences in seedling vigour both between species and between varieties. [12]

A laboratory seed test for germination should be carried out before seeding to calculate seeding rates. However, a simple preliminary test on-farm can be done in soil after harvest or during storage. Results from a laboratory germination and vigour test should be used in seeding rate calculations.

Grower retained seed should be tested for germination and vigour at least twice:

- Immediately post-harvest, to assess if the seed is worth retaining or better sold for grain or fed to livestock.
- Just before sowing, after grading and treatment with fungicide seed dressing, so seeding rates can be adjusted to compensate for any decline in quality during storage over summer.

Weather-damaged seed deteriorates quicker than usual in storage and a third test in mid-storage is advised. This will indicate if the seed is still suitable for sowing and if not, allows time to consider other options.

Pulse seed should have a minimum germination of 80% to be kept for sowing. Seeding rates for seed with only 50% germination should not be used.

Whilst increasing the seeding rate of poor quality seed may seem reasonable, it carries a high risk of seedling disease. If the poor quality is caused by seed-borne pathogens, the seed will be a source of infection for healthy seed and seedlings.

Simple germination tests are best done under cooler conditions. For beans, chickpea, lupins, peas and vetch, the sample size required is 1 kg for each 25 t of seed. There are two quick methods that both require 100 seeds for the test:

- Method 1: Place 100 seeds between at least four sheets of paper towel, roll up, fold the ends over and soak in fresh water for 30 minutes. Drain, put the 'seed doll' in a tray and place on the kitchen bench or workshop table. Ensure 'seed doll' does not dry out. After three, four, and five days unwrap 'doll', remove germinated seeds, taking a note of their vigour and re-wrap the 'doll'. After the fifth day, count the non-germinated and mouldy seeds.
- Method 2: Fill a flat shallow (5 cm deep) garden tray or non-rusty baking tray with clean sand, potting mix or freely draining soil. On the soil surface arrange your 100 sends in 10 rows and push the seeds into the soil with a pencil marked to a depth of 3 cm (Photo 3 - right). Cover with a little more sand/soil and water

11 J Kamboozia, G McDonald, H Reimers. (1993). The effect of seed size, seed protein and genotype on seedling vigour in some grain legumes. 7th Aust. Agron. Conf.©, SARDI.

12 J Kamboozia, G McDonald, H Reimers. (1993). The effect of seed size, seed protein and genotype on seedling vigour in some grain legumes. 7th Aust. Agron. Conf.©, SARDI.

gently. Keep soil moist but not wet as overwatering will result in fungal growth and possible rotting.

A germination test does not always accurately predict emergence. For a valuable crop like chickpeas a laboratory test should be used. Growers can also conduct their own emergence test, as per method 2. If this is conducted in the intended paddock this will also help to identify potential herbicide residue problems.

After 7–10 days the majority of viable seeds will have emerged (Photo 3 - left). Count only the normal and healthy seedlings. If left for another few days you will also be able to assess how many of the germinated seeds are actually deformed (e.g. missing cotyledons or mould/disease affected), which will also give some indication of vigour. [13]

Photo 3: *Doing your own germination test (left). Seedling growth after 12 days and after 5 days (right)*

Photo: E. Leonard, Kevin Moore. Source: Pulse Australia.

Sampling should be random and numerous subsamples should be taken to give best results. Sample either while seed is being moved out of the seed cleaner, storage or truck, or otherwise from numerous bags. Do not sample from within a silo or a bagging chute, as it is difficult to obtain a representative sample and is dangerous. Failure to sample correctly or test your seed could result in poor establishment in the field.

If an issue is suspected with kept grain, then it is better to get a sample tested early to avoid the cost of grading and time lost to procure suitable seed.

Testing seed vigour

Seed vigour is more accurately assessed by commercial laboratories as the assessment method is both temperature and time based. The seedling vigour test will also provide a better indication of the number of abnormal seeds present in addition to the germination percentage.

In years of drought or a wet harvest, seed germination can be affected, but also more importantly, seedling vigour can be reduced. Poor seedling vigour can heavily affect establishment and early seedling growth. This can often occur under more difficult establishment conditions such as deep sowing, crusting, compaction, wet soils, or when seed treatments have been applied. Some laboratories also offer a seed vigour test when doing their germination testing. Vigour represents the rapid, uniform emergence and development of normal seedlings under a wide range of conditions. Several tests are used by seed laboratories to establish seed and seedling vigour.

Cool germination and cold tests

A cool or cold test is done to evaluate the emergence of a seed lot in cold wet soils, which can cause poor field performance. The cold test simulates adverse field conditions and measures the ability of seeds to emerge. It is the most widely used vigour test for many crops. It is also one of the oldest vigour tests.

This test is used to:

13 L Jenkins, K Moore, G Cumming. Pulse Australia. Chickpea: High Quality seed. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/high-quality-seed

- evaluate fungicide efficacy
- evaluate physiological deterioration resulting from prolonged or adverse storage, freezing injury, immaturity, injury from drying or other causes
- measure the effect of mechanical damage on germination in cold, wet soil
- provide a basis for adjusting seeding rates.

This test usually places the seed in cold temperatures (5–10°C, with variations between laboratories) for a period, which is then followed by a period of growth. Then the seed is evaluated relative to normal seedlings according to a germination test. Some laboratories also categorise the seedlings further into vigour categories and report both of these numbers.

Accelerated ageing vigour test

Accelerated ageing estimates longevity of seed in storage. It is now also used as an indicator of seed vigour and has been successfully related to field emergence and stand establishment. This tests seed under conditions of high moisture and humidity. Seeds with high vigour withstand these stresses and deteriorate at a slower rate than those with poorer vigour. Results are reported as a percentage, and the closer the accelerated ageing number is to the germination result, the better the vigour. Results are expressed as a percentage of normal germination after ageing (vigorous seedlings).

Conductivity vigour test

The conductivity test measures electrolyte leakage from plant tissues and is one of two ISTA-recommended vigour tests. Conductivity test results are used to rank vigour lots by vigour level. It is important to have a germination test done too, because a conductivity test cannot always pick up all chemical and pathogen scenarios, which may be seed-borne.

Tetrazolium test (TZ) as a vigour test

The tetrazolium test is used to test seed viability, but is also useful as a rapid estimation of vigour of viable seeds. It is done in the same way as a germination test, but viable seeds are evaluated more critically into categories of:

- High Vigour—staining is uniform and even, tissue is firm and bright.
- Medium Vigour—embryo is completely stained or embryonic axis stained in dicots. Extremities may be unstained. Some over-stained or less firm areas exist.
- Low Vigour—large areas of non-essential structures unstained. Extreme tip of radicle unstained in dicots. Tissue is milky, flaccid, and over-stained.

Results have shown good relationships with field performance, and the test is useful for pulses.

Weed contamination testing

Sowing seed free of weeds cuts the risk of introducing new weeds. It also reduces the pressure on herbicides, especially with increasing herbicide resistance. Tests for purity of a seed sample can be conducted if requested, including the amount of weed seed contamination. Do not plant seed contaminated with weed seeds.

Disease testing

Seed-borne diseases such as ascochyta blight pose a serious threat to yields. Seed-borne diseases can strike early in the growth of the crop when seedlings are most vulnerable and result in severe plant losses and hence lower yields. Testing seed before sowing will identify the presence of disease and allow steps to be taken to reduce the disease risk. If disease is detected, the seed may be treated with a fungicide before sowing or a clean seed source may be used.

2.2.4 Seed purity

Accurate identification of chickpea varieties is critical to Ascochyta blight management in commercial crops.

Australian chickpea varieties differ in their reaction to Ascochyta blight. Varieties released before 2005 are susceptible to Ascochyta blight and, in seasons conducive to disease, require intensive management with foliar fungicides. Most cultivars released in 2005 and later have improved Ascochyta blight resistance and require fewer fungicide sprays. However, a new strain of ascocychta blight has left previously resistant varieties susceptible to the disease.

Contamination between seed varieties has been found to lead to higher than expected disease susceptibility in moderately resistant crops. Contamination or a mix-up in source of planting seed might account for the observed differences in Ascochyta blight levels in chickpea crops grown from grower-retained seed. This is part of the large issue of maintaining genetic purity in Australian chickpea varieties after their release.

Key findings from chickpea seed purity research:

- DNA evidence has identified genetic contamination in commercial chickpea crops going back to at least 2011.
- Crop inspections have revealed obvious differences among plantings believed by growers to be the one variety.
- Minimise the risk of contamination of your 2014 planting seed by obtaining seed from a registered seed merchant.
- When retaining your own seed, put in place a quality control system to avoid accidental contamination.

The extent of purity contamination is not yet determined; however, testing results from 36 seed lots suggest that the seed purity problem is far bigger than currently thought. Although the problem first surfaced in 2011, pathologists say it appears to be getting worse (Photo 4).

For details on disease ratings for chickpea varieties, see Section 9 - Diseases, Table 3.

Photo 4: *As the issue of seed purity increases, growers should treat crops from suspect seed as a susceptible variety.*

Photo: Rachel Bowman, Seedbed Media

Accurate identification of chickpea variety is essential for:

- implementing appropriate disease-management strategies
- minimising the risk to resistance genes in moderately resistant varieties from increased inoculum generated on contaminant plants or 'mix-up' crops of susceptible varieties
- maximising marketing opportunities by producing pure seed of one variety
- supporting growers' legal rights (e.g. if seed you purchased is not what you paid for)
- assessing compliance with plant breeders' rights legislation, thus ensuring breeding programs receive the appropriate royalties
- prolonging the commercial life of new varieties
- providing confidence in the chickpea seed industry
- providing technical support to research programs (e.g. knowing the genotype of a plant from which an isolate is obtained is critical to the current GRDC project on the variability of the Australian population of the chickpea Ascochyta blight pathogen).

An example consequence of varietal impurity - Cost of Ascochyta blight management

In a season that is conducive to chickpea Ascochyta, Tamworth-based NSW DPI research has shown that a crop of pure PBA HatTrick⁽ᶲ⁾ will require two foliar fungicide sprays totalling $30/ha. A crop of an Ascochyta blight-susceptible variety, e.g. Jimbour, would need six sprays costing $90/ha. This equates to a difference of $30,000 for a 500-ha planting. If you are unsure of the variety's identity or it is a mixture, the crop must be treated as a susceptible variety. [14]

2.2.5 Seed storage

Storing pulses successfully requires a balance between ideal harvest and storage conditions. Harvesting at 14% moisture content captures grain quality and reduces mechanical damage to the seed but requires careful management to avoid deterioration during storage.

Tips for storing pulses:

- Pulses stored at >12% moisture content require aeration cooling to maintain quality.
- Meticulous hygiene and aeration cooling are the first lines of defence against pest incursion.
- Fumigation is the only option available to control pests in stored pulses, and requires a gas-tight, sealable storage.
- Avoiding mechanical damage to pulse seeds will maintain market quality, seed viability, and be less attractive to insect pests. [15]

Most pulse seed should only be stored for 12 months, although longer storage periods are possible with high quality seed, provided both grain moisture and temperature within the silo can be controlled. Rapid deterioration of grain quality occurs under conditions of high temperature/moisture and with poor seed quality including weathered, cracked, and diseased seed.

Ideally, chickpea needs to be stored at 13% moisture content and at temperatures below 30°C. Storage at very low moisture contents (<10%) may make chickpeas (particularly Kabuli types) more susceptible to damage during subsequent handling, as the seed shrinks away from the seed coat.

VIDEOS

2. Chickpea seed quality – Kevin Moore, 2013.

14 K Moore, K Hobson, A Rehman, J Thelander (2014) Chickpea varietal purity and implications for disease management. GRDC Update Papers 5 March 2014, http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Chickpea-varietal-purity-and-implications-for-disease-management

15 GRDC (2012) Storing pulses. GRDC Grain Storage Fact Sheet March 2012, www.grdc.com.au/GRDC-FS-GrainStorage-StoringPulses

VIDEOS

3. Over the Fence: Insure seed viability with aerated storage.

MORE INFORMATION

Storing pulses

Like other grain, chickpea seed quality deteriorates in storage. Most rapid deterioration occurs under conditions of high temperature and moisture. Crops grown from seed that has been stored under such conditions may have poor germination and emergence.

Reducing temperature in storage facilities (to below 30°C) is the easiest method of increasing seed longevity. Temperatures can be reduced in grain silos by painting the outside of the silo white (temperatures reduced by 4–5°C), and/or aerating silos with dry, ambient air.

2.3 Reducing temperature in grain silos

- Paint the outside of the silo with white paint. This reduces storage temperature by as much as 4–5°C and can double safe storage life of grains.
- Aerate silos with dry, ambient air. This option is more expensive, but in addition to reducing storage temperatures, is also effective in reducing moisture of seed harvested at high moisture content.
- Heat drying of chickpea sowing seed should be limited to temperatures ≤40°C. [16]

Insect pests in storage

Insects are generally not considered to be a major problem in stored chickpea seed. However, where a prior infestation exists in the storage structure (most commonly as a result of cereal grain residue) then the infestation can develop and spread in the chickpeas. Ensure all handling equipment and storages are cleaned of old cereal grain before they are used to handle chickpeas. Good hygiene, combined with aeration cooling, should prevent infestations developing. If insect pests are found in stored chickpeas, the only registered treatment is phosphine fumigation.

For more information, See Section 13: Storage.

2.3.1 Safe rates of fertiliser sown with the seed

All pulses can be affected by fertiliser toxicity. Higher rates of phosphorus (P) fertiliser can be toxic to establishment and nodulation if drilled in direct contact with the seed at sowing. Practices involving drilling 10 kg/ha of P with the seed at 18-cm row spacing through 10 cm points rarely caused any problems. However, with the changes in sowing techniques to narrow sowing points, minimal soil disturbance, wider row spacing, and increased rates of fertiliser (all of which concentrate the fertiliser near the seed in the seeding furrow), the risk of toxicity is higher. Agronomists, however, can present anecdotal reports where toxicity has not been a problem.

The effects are also increased in highly acidic soils, sandy soils, and where moisture conditions at sowing are marginal. Drilling concentrated fertilisers to reduce the product rate per hectare does not reduce the risk.

The use of starter nitrogen (N), e.g. DAP, banded with the seed when sowing pulse crops has the potential to reduce establishment and nodulation if higher rates are used. On sands, up to 10 kg/ha of N at 18-cm row spacing can be safely used. On clay soils, do not exceed 20 kg/ha of N at 18-cm row spacing.

Deep banding of fertiliser is an option, as well as broadcasting and incorporating drilling, pre-seeding, or splitting fertiliser applications so that lower rates or no P is in contact with the seed. [17]

16 L Jenkins, K Moore, G Cumming. Pulse Australia. Chickpea: High Quality seed. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/high-quality-seed

17 GRDC (2008) Grain Legume Handbook update 7 Feb 2008. Grain Legume Handbook Committee, supported by the Grains Research and Development Corporation (GRDC), http://elibrary.grdc.com.au/project/glh00002

2.4 Future breeding directions

The current PBA program continues to focus on regional adaptation, higher grain yields, and greater levels of varietal resistance to the main two chickpea diseases of Ascochyta blight and Phytophthora root rot.

Additional valuable traits that the breeding program is working with include:

- resistance to Botrytis grey mould
- virus resistance
- improved resistance to root-lesion nematodes (*Pratylenchus thornei* and *P. neglectus*)
- improved tolerance to soil salt levels
- improved reproductive cold tolerance. [18]

18 G Cumming (2014) Chickpea varieties selecting horses for courses. GRDC Update Papers 5 March 2014 https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Chickpea-varieties-selecting-horses-for-courses

Planting

Key messages

- The strain of rhizobia used for inoculating chickpeas is highly specific (Group N, CC1192). Inoculation is essential for effective nodulation and will result in a crop that is self-sufficient for N and provide soil health benefits in subsequent seasons.
- All seed, regardless of source, should be treated with a registered thiram-based fungicide seed dressing prior to sowing.
- The sowing window in many favourable areas tends to be after cereals and other pulses. Early sowing (even dry sowing) is common in the lower rainfall areas such as the Mallee.
- While yields are relatively stable within the range of 35–50 plants/m^2, higher seeding rates (50 plants/m^2) produce the highest yields in western and southern areas.
- Sowing at 30–50 cm spacing is becoming common. Some innovators are sowing in 50–100 cm row spacing into standing cereal stubble and using inter-row spraying for weed control. Know the advantages and disadvantages of row spacing.
- Sow chickpeas 5–7 cm deep into good moisture. The seedlings are robust, provided high quality seed is used. There are also benefits to deep-planting chickpea.

MORE INFORMATION

Checklist for Chickpea growers in the Southern region.

3.1 Inoculation

Pulses, including chickpea, have the ability to 'fix' their own nitrogen from the air via nodules on their roots if specific nitrogen-fixing bacteria (rhizobia) are available. Grain legume crops (such as pulses) and pasture legumes initiate a symbiotic relationship with rhizobia bacteria, to form nitrogen-fixing root nodules. The chickpea is an introduced crop to Australia and, as such, seeds must be treated (inoculated) with the correct strain of rhizobia (symbiotic N-fixing bacteria) before planting.

In South Australia, all chickpea seed sown should be inoculated with the specific chickpea rhizobium inoculum (Group N), even in paddocks where chickpeas have been grown before. If the seed is to be treated with an insecticide or fungicide, it should be inoculated last, immediately before sowing. In Victoria, on acid soils, it is essential to inoculate every sowing.

In the Wimmera, inoculate seed with chickpea Rhizobium (Group N inoculant) if the paddock has never grown chickpeas or only one 'successfully' nodulated crop. Effective nodulation is 70% or more plants with healthy nodules.

Chickpea rhizobia can survive in the soil for up to eight years. The sure way to successful nodulation is to inoculate every time, but on Wimmera grey clay soils the following compromise works:

1. Grow two well-inoculated crops of chickpeas preferably four years apart.
2. If up to eight-year break from inoculation: chickpeas grown in this period do not need inoculation.
3. If it is more than eight years since chickpea inoculation, inoculate next crop of chickpeas.
4. Repeat steps 2 and 3.

The strain of rhizobia used for chickpeas is highly specific (Group N, CC1192). Inoculation is essential for effective nodulation and will result in a crop that is self-sufficient for N and provide soil health benefits in subsequent seasons. This nitrogen (N) fixation process has a national benefit of close to $4 billion annually in Australian cropping systems. Growers can treat seed prior to sowing with inoculants containing

VIDEOS

1. GCTV13: Legumes – Sowing preparation.

2. GCTV13: Legumes – Inoculant compatibility.

MORE INFORMATION

Inoculating pulse crops

live bacteria to stimulate nodulation and the N-fixation capacity of legumes. It is important to match the correct inoculant group to each legume. [1] The Group N bacteria are regarded as an 'aggressive nodulator'. This effectively means that nodulation will be successful in meeting the crop's nitrogen requirements, provided the inoculants are handled and stored in a manner that will ensure bacterial survival and that growers adopt effective inoculation practices on-farm.

The most common method of inoculating chickpea is to coat the seed with a slurry of peat-based inoculum immediately before planting (Photo 1). It is important to treat only the seed that can be planted the same day. Exposure to drying winds, high temperatures, or direct sunlight will rapidly kill the bacteria. [2]

Use of granular inoculant is becoming more popular, particularly with early sowing into dry soil before germinating rains. Liquid injection of inoculant into the seed row is also becoming more widely used.

Photo 1: *Forms of rhizobia (left to right): Easyrihiz freeze-dried, Nodulator granules, Alosca granules, N-Prove granules and peat inoculant.*

Photo: M. Denton, DPI Vic.

Only purchase refrigerated (but not frozen) inoculum from a reputable supplier and then store it in a cool, dry place. For maximum survival, peat inoculant should be stored in a refrigerator at ~4°C until used. If refrigeration is not possible, store in a cool, dry place away from direct sunlight. Granules and other forms also need to be stored in a cool place out of direct sunlight. Do not store an opened inoculum packet, as it will deteriorate rapidly. Discard the inoculant after the expiry date, because the rhizobia population may have dropped to an unacceptable level. Treat seed within 24 hours of sowing and sow into moist soil. Consider new technologies that are now also available and may suit your operations, e.g. freeze-dried inoculums, water liquid injection, granular inoculums. Dry sowing of chickpeas is now possible if using granular inoculums that enable rhizobia survival until rain arrives to germinate seed. [3]

Inoculated seed must be planted into moisture within 12 hours of treatment. The sooner the better, as fungicide seed dressings can effect survival of the bacteria. Inoculated seed and acidic fertilisers should not be sown down the same tube. The acidity of some fertilisers will kill a high proportion of the rhizobia and render inoculation ineffective. Neutralised (e.g. Super lime) and alkaline fertilisers (e.g. DAP, Starter NP, lime) can be safely used. [4]

1 GRDC. 17/08/16 Media Releases. Growers get the nod to check legumes for nitrogen fixation. www.grdc.com.au/media-news

2 DAFF (2012) Planting chickpeas. Department of Agriculture, Fisheries and Forestry August 2012, http://www.daff.qld.gov.au/plants/field-crops-and-pastures/broadacre-field-crops/chickpeas/planting

3 Pulse Australia. Southern Pulse Bulletin PA 2010 #05 – Chickpea checklist for southern growers. http://www.pulseaus.com.au/storage/app/media/crops/2010_SPB-Chickpea-checklist-south.pdf

4 Pulses Australia. Chickpea Production: Southern and Western region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

IN FOCUS

Nutrient uptake and yield of chickpea (*Cicer arietinum* L.) inoculated with plant growth- promoting rhizobacteria

Application of every inoculation treatment studied in this research, especially treatments which contained *Azospirillum* or *Azotobacter*, may stimulate growth and yield of chickpea as compared with uninoculated plants.

Inoculating legumes with rhizobia can achieve substantial increases in legume nodulation, grain and biomass yield, nitrogen fixation and post-crop soil nitrate levels. Plant growth promoting rhizobacteria (PGPR) represent a wide variety of soil bacteria which, when grown in association with a host plant, result in stimulation of growth of their host plant. Several mechanisms have been suggested by which PGPR can promote plant growth, including phytohormone production, N_2 fixation, stimulation of nutrient uptake, and biocontrol of pathogenic microorganisms.

This research was carried out to evaluate the effects of single and combined inoculation with plant growth-promoting rhizobacteria from four genera including *Azospirillum*, *Azotobacter*, *Mesorhizobium*, and *Pseudomonas* on nutrient uptake, growth and yield of chickpea plants under field conditions.

Nodulation and nutrient concentration in shoots were significantly affected by the treatments at the beginning of flowering stage. The maximum dry weight of root nodules was recorded by applying the combined inoculation with *Azospirillum* spp. + *Azotobacter chroococcum* 5 + *Mesorhizobium ciceri* SWRI7 + *Pseudomonas fluorescens* P21. All inoculants were statistically superior over uninoculated control with respect to nitrogen concentration of shoots.

The treatments containing *Azospirillum* + *Azotobacter* significantly improved phosphorus concentration in shoots. Grain yield, biomass dry weight and nitrogen and phosphorus uptake of grains were statistically improved by applying every inoculation treatment in comparison with control plants. Group comparisons between treatments showed that the occurrence of *Azospirillum* or *Azotobacter* inoculants in the treatment composition caused an expressive improvement in grain yield and plant biomass. [5]

3.1.1 Inoculation checklist

Important points when purchasing and using inoculants:

- Check the expiry date on packet.
- Packets should be stored at around 4°C.
- Do not freeze (below 0°C) or exceed 15°C.
- Use Group N chickpea inoculum.
- Prepare slurry and apply in the shade, avoiding exposure to high temperatures (>30°C), direct sunlight, and hot winds.

5 Rokhzadi, A., & Toashih, V. (2011). Nutrient uptake and yield of chickpea (*Cicer arietinum* L.) inoculated with plant growth-promoting rhizobacteria. Australian Journal of Crop Science, 5(1), 44.

- Accurately meter adhesive slurry onto the seed. Too much water means sticky seeds and blockages in the seeder.
- Avoid high-speed mixing in augers and inoculate at the top of the auger not the bottom.
- Sow inoculated seed immediately. Never delay more than 12 hours.
- Check air-seeders for excessively high temperatures in the air stream. Temperatures >50°C will kill the rhizobia. [6]

3.1.2 Inoculant types

Peat inoculum

The traditional method of supplying rhizobia to seed is with peat inoculum. Read the label and apply the inoculant according to the directions. Most pulse crop inoculums for application to seed contain a pre-mixed sticker which helps adhesion to the seed. Be cautious and read the inoculant label regarding adding any approved insecticides, fungicides, herbicides, detergents, or fertilisers into the slurry as these may be toxic to the rhizobia. Inoculated seed should be used within 24 hours when applied alone, and within four hours is applied in conjunction with a fungicide. This is freeze dried to be applied either on seed or in furrow via water injection.

In-furrow water injection

Water or fluid injection of inoculants into the seed row is becoming more common as it has been adapted to pulse growing and modern machinery. It can be used where machines are set up for liquid N on cereals, and where fungicides are used to treat seeds before sowing. Water injection of inoculant requires at least 80 L of water per hectare and is better with more.

The slurry-water solution is sprayed under low pressure into the soil in the seed row during seeding. Benefits of the new inoculant carrier types over freeze-dried peat are that they are convenient, 'dissolve' more readily, and do not have the requirement for filtering out peat hairs etc. Read the label before mixing any fungicides.

Granular forms

Granular inoculants are applied like fertiliser as a solid in the seed furrow or near to the seed, and avoid many of the compatibility problems that rhizobia have with fertilisers and fungicides. They also eliminate the application procedures needed with peat inoculum. Granular inoculants also offer some advantages when dry sowing.

Granules can vary and, depending on the product, may be dry or moist, uniform, variable, powdery, coarse, or fine.

Granules contain fewer rhizobia per gram than peat based inoculants, so they must be applied at higher application rates. The size, form, uniformity, moisture content, and rate of application of granules differ between products. Depending on the product or row spacing sown, application rates can vary from 4 kg/ha to 10 kg/ha to deliver adequate levels of nodulation.

Several granular inoculant formulations are now available, and they are not all the same in composition, practicalities of use, and performance. Seek independent trial information for your area when making comparisons between products.

ALOSCA is a dry clay (Bentonite) granule. BASF produces moist clay granules (Nodulator), while Novozymes Australia also produces a moist granular product.

Even application is best achieved through a dedicated third box on the seeder (Photo 2). Maintaining an even application rate when mixed with the seed is difficult as the granules shake down in the seed lot. Achieving an even distribution rate in the fertiliser box is also difficult, and rhizobia survival becomes an important consideration. Only ALOSCA claim compatibility when mixed with fertiliser. [7]

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6 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

7 Pulse Australia. Pulse inoculation techniques. http://www.pulseaus.com.au/growing-pulses/publications/pulse-inoculation

Photo 2: *An 'after-market' third box fitted to a Flexicoil box to enable application of granular inoculums. Note that granular inoculums cannot be applied mixed with the seed (uneven distribution of seed and/or inoculums occurs). Rhizobial survival is severely jeopardised if granular inoculums are applied mixed with fertiliser.*

Source: Grain Legume Handbook, 2008

3.1.3 Choosing an inoculant type

All types of inoculants (Photo 1) will result in a well-nodulated crop in good conditions (Table 1). The choice of inoculant type by growers will depend on:

- experience
- paddock history, i.e. the need for added rhizobium
- product availability
- relative cost
- perceived efficacy
- ease of use
- the suitability of machinery to deliver the inoculum.

When conditions are less than ideal, making the right choice becomes more critical.

The rhizobia bacteria need moisture to survive. When contained in the carrier, i.e. the peat material or the granule form, they will survive for up to 12 months when stored well. Read the expiry date before use.

However, once applied, the survival rate is highly dependent on available soil moisture. This particularly applies to inoculum applied to the seed or to the soil as slurry. Dry soil conditions after sowing will kill off the rhizobia. Moisture will be needed within 2–3 days after sowing to maintain adequate numbers. If introduced rhizobia are essential for crop health, dry sowing should be avoided and caution should be used if sowing into a drying seedbed with a poor forecast for follow-up rain.

Granules by comparison are ideally suited to maintaining rhizobia numbers in dry soil for extended periods before rain arrives. The rhizobium is maintained within the granule which continues to protect it until the soil wets and the rhizobia can start multiplying. They are ideal to use if dry sowing is being considered. Additionally they enable fungicides, which may be toxic to the rhizobia, to be applied to the seed without causing a reduction in rhizobia numbers. [8]

Table 1: *Inoculant types available.*

Inoculant product	Application method
BASF (NODULAID™NT)	Peat inoculant, applied as a slurry/powder/liquid to the seed or in furrow to the soil
BASF (NODULATOR™)	Clay granular inoculant to be applied in furrow to the soil
New-Edge Microbials (EasyRhiz™)	Freeze-dried inoculant, made up into a liquid and applied to the seed or in furrow by water injection into the soil
New-Edge Microbials (Nodule N)	Peat inoculant, applied as a slurry to the seed
ALOSCA Technologies	Dry clay (bentonite) granular inoculant, applied in furrow to the soil
Novozymes Australia (Cell-Tech™)	Peat inoculant. Applied as a slurry/powder/liquid to the seed or in furrow to the soil
Novozymes Australia (Tag-Team™)	Peat inoculant with phosphate-mobilising soil fungi Penicillium bilaii.

Source: Pulse Australia.

3.1.4 When to inoculate

If crops within an inoculum group have not previously been grown in the paddock to be sown, then seed of the crop should be inoculated immediately prior to sowing; otherwise, a nodulation failure may occur.

If conditions for nodulation are likely to be adverse; i.e. waterlogged, acid soils, or lighter soils, then it may help to use some starter N, e.g. mono- or di-ammonium phosphate (MAP or DAP). This will stimulate early root growth until the numbers of naturally occurring rhizobia build up and begin fixing N. [9]

Rhizobia will persist in the soil and, depending on a range of conditions, can inoculate a subsequent pulse. If the paddock has previously grown the same pulse, the number of rhizobia remaining in the soil will be affected by the:

- time since the pulse was last grown
- health of the crop
- type of rhizobia
- soil pH and texture.

Rhizobia types vary in their ability to persist in the soil until the host pulse crop is regrown. Less is known of the survivability of chickpea rhizobia (Group N). Inoculating at sowing is recommended, regardless of other considerations.

Lupin rhizobia (Group G) are most resilient and survive very well in low pH (down to pH 5) sandy soils. Pea and bean rhizobia (Groups E & F) survive well in neutral to alkaline soils with good texture (loams or clays).

If a well-nodulated lupin or pea crop has been grown in the previous four years, a response to inoculation is less likely. However, pea and bean rhizobia survive poorly in low pH or sandy soils. The safest option is to inoculate the crop, especially on light textured soils.

The cost of inoculation is low and worth the effort if there is any doubt about the viability of residual soil rhizobia.

8 Pulse Australia. Pulse inoculation techniques. http://www.pulseaus.com.au/growing-pulses/publications/pulse-inoculation

9 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

A benefit of inoculating a crop where rhizobia already exist is that an improved strain will be introduced which could result in better persistence for future pulse crops. Research continues to find more robust and efficient rhizobia strains for all pulse species. The strain used today in any group will be more advanced than those introduced to a paddock in the past. [10]

3.1.5 Inoculum survival

Moist peat provides protection and energy while the unopened packet is being stored. Inoculated seed should be sown directly into moist soil. Rhizobia can dry out and lose viability once applied to seed and not in moist soil. Granular inoculant forms may not dry out as quickly.

Most peat inoculants now contain an adhesive, which delays drying and increases survival of the rhizobia. Use a peat slurry mixture within 24 hours. Sow seed inoculated with peat slurry as soon as possible, but certainly within 12 hours, being sure to keep the seed in a cool place, away from sunlight.

With non-peat based inoculants, such as freeze-dried rhizobia, it is recommended that treated seed be sown within five hours of inoculation. The rhizobia survive for longer in granules than when applied on seed. Hence, when dry sowing pulses, granular inoculant is preferred over peat and liquid injection methods.

Dry-dusting the peat inoculant into the seed box is not an effective means of distributing or retaining rhizobia uniformly on seed. Under some conditions, rhizobial death is so rapid where dry dusting is used that no rhizobia remain alive by the time the seed reaches the soil. [11]

3.1.6 Inoculant quality assurance

Legume inoculants sold to Australian farmers must pass a rigorous quality assurance (QA) program. Cultures of inoculant are tested by the Australian Legume Inoculants Research Unit (ALIRU) to establish that the correct rhizobial strain is present and the viable cell number exceeds a minimum value (Table 2).

Table 2: *ALIRU Quality Assurance rhizobia minimum numbers.*

Product	Viable rhizobia (no./g)	Rate per ha	Rhizobia (no./ha)	Expiry (months)
Peat	1×10^9	250 g	3×10^{11}	12–18
Liquid	5×10^9	300 mL	2×10^{12}	6
Granular	1×10^7	10 kg	1×10^{11}	6
Freeze-dried	1×10^{12}	0.15 g	2×10^{11}	24

Source: Grain Legume Handbook, 2008.

3.1.7 Inoculation methods

Pulses have historically been inoculated with rhizobia slurry onto the seed, but now rhizobia can be purchased in a form suitable to be applied with water injection into the soil, or as granules that are sown with the seed from a separate box. For water injection, the inoculant is mixed with water and applied at low pressure through tubes into each seed furrow. Using granules usually requires a third seed box as granules will shake out if mixed with seed and can lose viability if mixed with fertiliser (Table 3).

10 Pulse Australia. Pulse inoculation techniques. http://www.pulseaus.com.au/growing-pulses/publications/pulse-inoculation

11 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

Table 3: *Survival of different inoculant types with various application methods.*

Inoculant type	Where inoculant is applied	Survival in dry or drying soil*	Compatibility with seed-applied fungicide
Peat	Seed	Low	Some (check label)
Freeze-dried	Seed or in-furrow (water injection)	Very Low	Very Low
Granular	Seeding furrow of below seed	Yes	Yes
In-furrow water injection	Seeding furrow or below seed	Very low	Yes

*Survival will depend on duration of dry conditions and soil pH.
Source: Pulse Australia, 2013.

For Chickpeas:

- Peat formulation: as slurry to seed (most common) or in furrow
- Freeze-dried formulation: as slurry to seed or liquid in furrow
- Granular formulation: in furrow at sowing. [12]

3.1.8 Inoculum slurry

Most inoculants now contain a pre-mixed sticker. When mixing the slurry, do not use hot or chlorinated water. Add the appropriate amount of the inoculant group to the solution and stir quickly. Mix into a heavy paste with a small amount of water prior to adding to the main solution. Add the inoculant suspension (slurry) to the seed and mix thoroughly until all seeds are evenly covered.

How to apply slurry to the seed:

- in a cement mixer (practical for small lots only unless a cement truck is used)
- through an auger (Photo 3)
- through a tubulator

When applying via an auger, make sure the auger is turning as slowly as possible. Reduce the height of the auger to minimise the height of seed fall. Perhaps add a slide, e.g. tin, to the outlet end of the auger to stop seed falling and cracking. Meter the slurry in, according to the flow rate of the auger (remember 250 g packet per 100 kg seed). Too much water means sticky seed and blockage problems in the planter.

12 GRDC. (2013). Inoculating legumes: The Back Pocket Guide. https://grdc.com.au/GRDC-BPG-InoculatingLegumes

Photo 3: *Pumping a slurry of rhizobia inoculant into the auger to coat seed before sowing.*

Source: GRDC.

Applying the slurry through a tubulator is similar to applying through an auger, except that the tubulator reduces the risk of damaging the seed (Figure 1). Its mixing ability is not as effective as an auger.

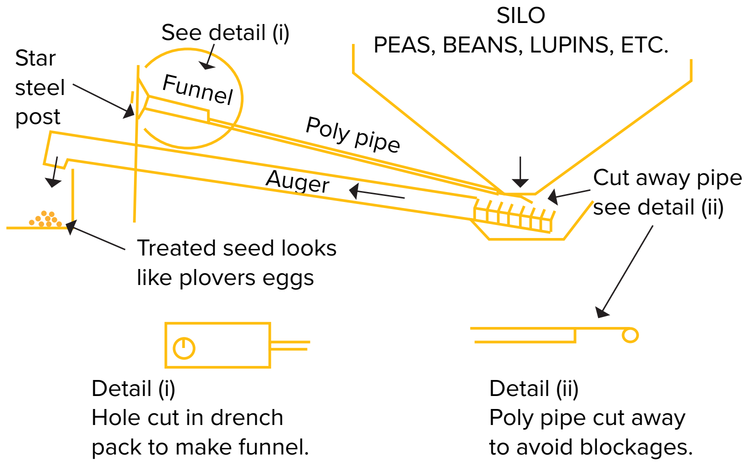


Figure 1: *Application of inoculum to seed through a tubulator.*

3.1.9 In furrow water injection

Injection of inoculants mixed in water is becoming more common. It can be used where machines are set up to apply other liquids at seeding, such as liquid N or phosphorus (P). Water injection of inoculant requires at least 40–50 L/ha of water, and is better with more water. The slurry–water solution is applied under low pressure into the soil in the seed row during seeding. Benefits of the new inoculants over peat are that they mix more readily, and do not have the requirement for filtering out peat. Compatibility of the inoculant with trace elements is not yet known, but extreme caution is advised because water pH is critical, and trace element types, forms and products behave differently between products and inoculants groups.

Trials have consistently shown superior nodulation from water injection of inoculum (Figures 2 and 3). This is likely to be due to the larger numbers of live bacteria being delivered into the soil in close proximity to the seed.

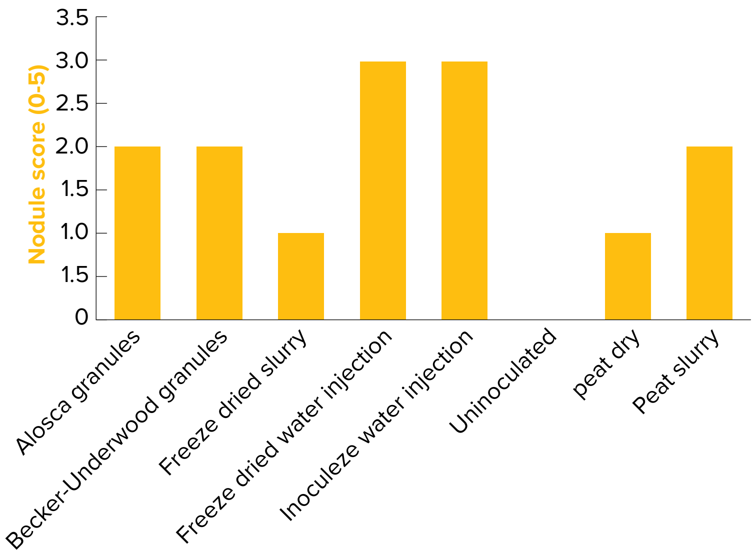


Figure 2: *Inoculation trial with nodule scores assessed at four weeks after planting.* [13]

Source: Pulse Australia

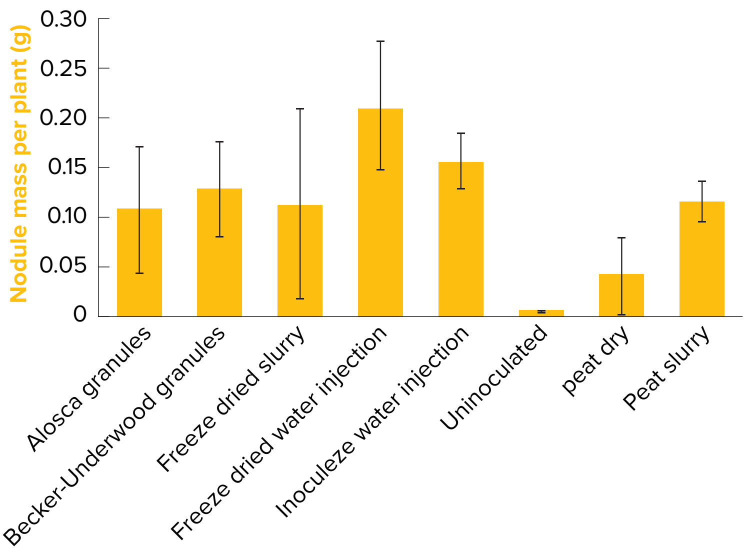


Figure 3: *Inoculation trial at Emerald (2005), nodule biomass at flowering.* [14]

Source: Pulse Australia

3.1.10 Inoculant application trials

Inoculation of chickpea seed with Group N rhizobia is recommended, regardless of paddock history. The standard method of mixing slurry and applying direct to seed still appears adequate; however, recent research has shown potential improvements

13 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

14 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

by injecting the rhizobia into the seed furrow with water as a carrier. Peat granules have on average performed as well as the standard slurry method, whereas attapulgite clay granules and bentonite clay granules have generally resulted in nodulation levels higher than the untreated control, but equal to or less than the standard slurry method.

Trials from 2008 to 2010 have compared the use of the available inoculant treatments. Figure 4 presents trial results in terms of nodule score by product type.

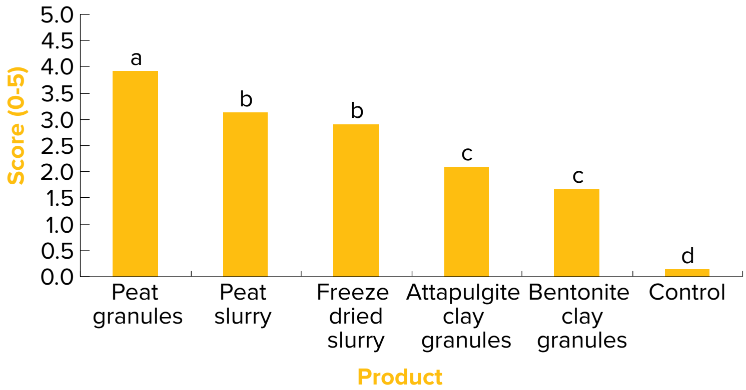


Figure 4: *Effect of inoculant treatment on nodulation of chickpea roots.* [15]

Limited trial data show that the most effective of the new technologies appears to be the application of rhizobia in-furrow with water (water-inject). This reduces the need to mix and apply slurry to the seed, but requires large volumes of water at sowing, as well as a liquid tank and plumbing to be incorporated into the seeder. Clay granules (attapulgite and bentonite) have often resulted in less nodulation than the standard slurry treatments. However, where chickpeas are a regular crop in the rotation, the reduced efficacy provided by the clay granules compared with the standard slurry treatment is likely to be less pronounced. Granules can reduce labour and downtime at sowing, so would only be recommended where real efficiency gains can be made. Peat granules resulted in nodulation levels greater than the clay granules in one of two trials. The use of standard slurry treatment (peat slurry) still appears to be a reliable method of application. In some cases, nodulation may be less than with the 'water inject', but this needs to be balanced with the extra machinery cost of liquid injection.

In one trial, nodulation from the slurry applied to seed method was significantly affected by fungicide (thiram + thiabendazole), where the fungicide and slurry were applied within one hour of each other. In the trials where the fungicide did not affect nodulation, the seed had been treated with fungicide at least several days before inoculation. In the fungicide-affected trial, the freeze-dried slurry treatment showed a greater reduction in nodulation from fungicide than what was seen from the peat slurry treatment.

For growers planting small areas of chickpeas, or who are content with current treatments methods, the traditional method of peat slurry application still appears reliable. Where the requirement for N fixation is high, liquid injection may improve outcomes. Liquid injection (once set up on a machine) may also provide logistical benefits.

Where chickpeas have been a regular crop in the rotation, granules may provide adequate nodulation and give logistical benefits such as reduced labour requirement.

15 R Brill, G Price (2011) Chickpea inoculation trials 2008–10. GRDC Update Papers March 2011, http://www.nga.org.au/results-and-publications/download/67/grdc-update-papers-general-/rhizobia-innoculation-methods-inchickpeas/grdc-adviser-update-paper-goondiwindi-march-2011.pdf

Note: These results present only one year of data. To gain a full understanding of the individual treatments used, the trials need to be replicated over several seasons and as part of different farming systems. [16]

3.1.11 Compatibility with other major factors

Pesticides

Rhizobia are living organisms. As a general rule, pesticides are toxic to rhizobia.

Almost all pulses require a fungicide applied to the seed to provide protection during early growth against foliar diseases. Occasionally an insecticide may also be needed.

Peat inoculants are also applied to the seed, bringing together two largely incompatible products. Mixing inoculum with a pesticide for seed treatment is possible with some products. Read the inoculum label to check for compatibility. BASF claims compatibility between its peat inoculum and Rovral®. However, the seed must be sown within several hours into moist soil to avoid reducing rhizobia viability.

Applying the fungicide to the seed prior to the inoculum is a safer method to reduce the risk of rhizobia death. The fungicide can be applied at any time leading up to sowing. The inoculum is then applied immediately before sowing into moist soil. If in doubt, do not mix the inoculant and any pesticide.

Granular inoculants remove this risk because the rhizobia and the pesticide are not in contact. If you need to use a potentially toxic seed pesticide treatment, granular inoculant may be worth considering.

Always read the inoculant label or contact the manufacturer for up-to-date information on compatibility. [17]

Fungicides

Caution should be used when treating pulse seed with a fungicide. Some insecticide and seed treatments can also cause problems. Check the inoculant and chemical labels for compatibility of the inoculant and fungicide or insecticide seed treatments.

Effect of fungicidal seed dressings on inoculum survival

While fungicide seed dressings reduce the longevity of the N-fixing bacteria applied to the seed, the effect can be minimised by keeping the contact period to as short as possible (Table 4).

Inoculate fungicide-treated seed as close as possible to the time of sowing.

Re-inoculate if not planted within 12 hours of treatment.

16 R Brill, G Price (2011) Chickpea inoculation trials 2008–10. GRDC Update Papers March 2011, http://www.nga.org.au/results-and-publications/download/67/grdc-update-papers-general-/rhizobia-innoculation-methods-inchickpeas/grdc-adviser-update-paper-goondiwindi-march-2011.pdf

17 Pulse Australia. Pulse inoculation techniques. http://www.pulseaus.com.au/growing-pulses/publications/pulse-inoculation

Table 4: *Effects of fungicide seed dressings on plant growth and nodulation in chickpeas.*

Treatment	Fresh weight (g)			Height (cm)	Nodulation score
	Shoot	Root	Total		
Nil	106	142	248	47	1.0
Inoculum only	130	244	374	57	4.5
Inoculum + Thiram	103	182	285	55	1.8
Inoculum then Thiram	119	208	327	58	3.2
Thiram then inoculum	117	212	329	55	3.8
Inoculum + Apron	106	173	279	54	1.8
Inoculum then Arpon	114	207	321	59	3.3
Apron then inoculum	113	206	319	55	3.6
l.s.d (P = 0.05)	19	33	31	9	0.6

Source: Trevor Bretag, formerly DPI Victoria.

MORE INFORMATION

Inoculating legumes: a practical guide (GRDC)

Inoculating legumes: the back pocket guide (GRDC)

Rhizobia inoculants fact sheet (GRDC)

Trace elements

Rhizobia can be compatible with a few specific trace element formulations, but many are not compatible with rhizobial survival. Mixing inoculants with trace elements should only occur if the trace element formulation being used has been laboratory-tested against the rhizobial type being used.

Note the differences between inoculant types for a given trace element product, as well as differences between trace element products with a given inoculant.

3.1.12 Nodulation and nitrogen fixation

Different pulses need different strains of rhizobia, so are grouped into 'inoculation groups'. Unless the right strain is present in the soil or has been supplied by adding a commercial inoculant at seeding time, effective root nodulation will not take place and little if any N will be fixed. These effects are not always immediately obvious above ground.

Where the host legume plant is grown infrequently in the cropping rotation, re-inoculation can be beneficial. Use of a commercial inoculant will ensure that nodulation is prompt, that nodules are abundant and that the strain of rhizobia forming the nodules is effective at fixing N (Photo 4).

Photo 4: *Well-nodulated chickpea plants.*

Source: Flickr.

When the legume germinates, the rhizobia enter the plant's roots, multiply rapidly and form a nodule. Effective nodule formation and function for the all-important 'N fix' requires good growing conditions, the appropriate rhizobia and a host plant. Rotation lengths of 3–4 years are recommended between successive chickpea crops as a disease management strategy (i.e. Ascochyta blight). At this re-cropping interval, sufficient levels of surviving Group N rhizobia are unlikely for effective nodulation.

Nodules remain inactive until the soil nitrate supply is exhausted (ineffective nodules remain white inside due to the absence of leghaemoglobin). Effective N-fixing nodules on the other hand, are rusty red or pink inside (Photo 5).

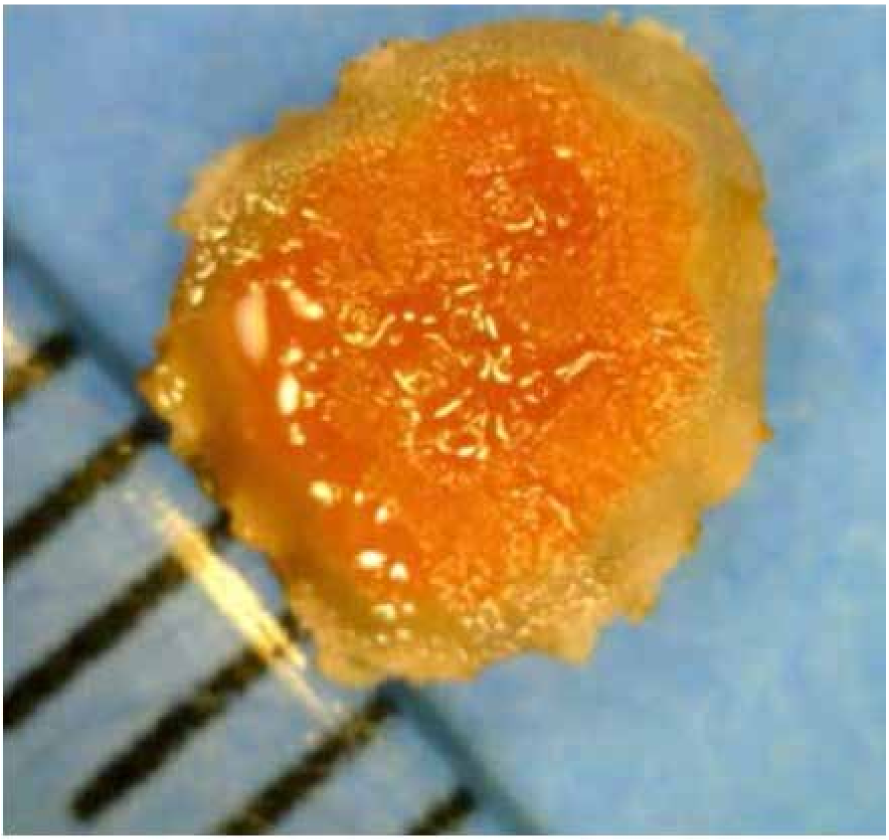


Photo 5: *Active nodules have a rusty red or pink centre.*

Photo: G. Cumming, Pulse Australia

3.1.13 Monitoring nodulation

Grain growers are encouraged to assess their legume crops for nodulation levels. Late winter and early spring is the best time to sample crops. It's important that growers who have used inoculants and also those who have not, check crops to see whether adequate nodulation is occurring. For farming systems to derive maximum benefits from legume N-fixation, optimal nodulation is necessary.

The best approach is to:

- Collect three samples of about 10 plants from three different spots within a paddock (suggested to be 20 m, 60 m and 100 m in from the edge), putting each sample of 10 plants in a separate bucket;
- Carefully wash off the soil in a bucket of water and rinse the roots to remove the remaining soil (soak for up to 30 minutes for heavy soil);
- Score each sample for the percentage of plants adequately nodulated and work out an average of scores for the three sampling locations.

Growers should look for:

- The number of nodules on a plant: the desirable number of nodules varies for different legumes (Photo 6), (for example 50 per plant is adequate for field pea, vetch and faba bean) and to some extent between soil types (lower numbers per plant are found on lighter soils);
- The location of nodules on the plant: where growers have inoculated seed, expect to see more nodules around the crown of the plant (where root meets shoot). These will boost the early growth of seedlings. Non-inoculated legumes will have nodules spread over the root system on crown, taproots, and laterals if rhizobia are already present in the soil;

- The colour inside the nodules: a red/pink colour means the nodules are effective and are fixing nitrogen (Photo 7). White or green nodules mean they are ineffective.

Photo 6: *Assessment of nodulation.*

Photos A. Gibson. Source: GRDC.

Key to chickpea root nodulation (above)

Score 0: taproot, absent; lateral, absent/few.

Score 1: taproot, few/medium; lateral, absent.

Score 2: taproot, medium; lateral, absent/low.

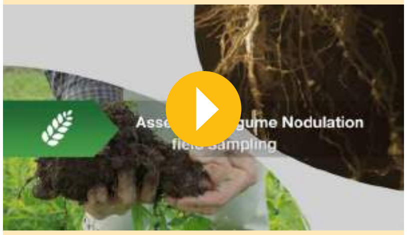
Score 3: tap root, medium/high; lateral, low.

Score 4: taproot, high; lateral, medium.

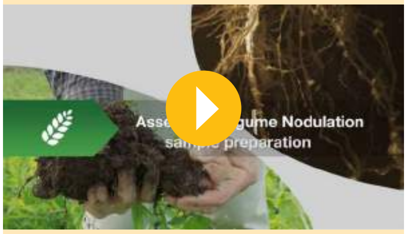
Score 5: taproot, high; lateral, medium.

VIDEOS

3. GCTV17: Legume nodulation — field sampling.



4. GCTV17: Legume nodulation — sample preparation.



5. GCTV17: Legume nodulation — sample scoring.

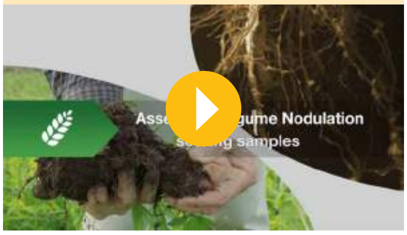


Photo 7: *A strong pink colour inside the nodules indicates that the rhizobia are actively fixing nitrogen.*

Photo: J Howleson, Pulse Australia.

If poor nodulation is apparent, growers should check their inoculation strategy to ensure best management practices are being followed. If both nodulation and plant performance are poor, reasons for poor nodulation need to be identified.

Poor nodulation can cause 10–50% yield loss in pulse crops, not to mention the lower potential nitrogen benefits to following crops. While a visual assessment will not indicate the actual level of nitrogen being fixed, which requires sophisticated scientific methods, looking at the roots to determine if there has been a nodulation failure or delay is worthwhile.

When looking at plants in the field, it is likely growers will find nodules on lateral roots as well as the main tap root. Look at all nodules when comparing the total nodule numbers to work out good or poor nodulation. Pulse crops that are poorly nodulated will be using more soil nitrogen than adequately nodulated crops, and fixing less nitrogen from the air.

Nodulation assessment will indicate whether this year's inoculation has been successful or whether troubleshooting is necessary. It will also tell a grower if non-inoculated legume should be inoculated next time it is grown in that paddock.

Poor nodulation can be caused by: no inoculation where low rhizobia numbers are present in soil; incorrect inoculant group or inoculant not being stored in cool conditions before use; inoculant effectiveness that has been reduced after mixing with certain types of seed dressings or liquid in furrow treatments (trace elements, pesticides, fertilisers or organic amendments); inoculated seed left for more than one day before sowing; and crop stress, such as nutrition, waterlogging, diseases or herbicides causing root damage. [18]

3.1.14 Use of nitrogen in inoculation

There has been some research into the effects of adding nitrogen to inoculated chickpea. One study applied inorganic nitrogen fertiliser at four levels (0, 50, 75, and 100 kg ha–1 (applied in urea form)) and two levels of inoculation with Rhizobium bacteria (with and without inoculation) as sub plots.

Application of N and Rhizobium inoculation continued to have positive effect on growth indices and yield components of chickpea. Lower levels of nitrogen

18 GRDC. 17/08/16 Media Releases. Growers get the nod to check legumes for nitrogen fixation. www.grdc.com.au/media-news

application and non-inoculated plants showed less growth indices including total dry matter (TDM), leaf area index (LAI), crop growth rate (CGR), relative growth rate (RGR) and net assimilation rate (NAR), while the highest values of these indices were observed at the high levels of nitrogen application and inoculated plants.

The highest plant height, number of primary and secondary branches, number of pods per plant, and number of grains per plant were obtained from the highest level of nitrogen fertiliser (100 kg urea ha–1) and Rhizobium inoculation.

Application of 75 and 100 kg urea ha–1 showed no significant difference in these traits. Moreover, the highest grain yield was recorded in the inoculated plants that were treated with 75 kg urea ha–1.

Based on the results of this study, the application of suitable amounts of nitrogen fertiliser (i.e. between 50 and 75 kg urea ha–1) as a starter can be beneficial in improving growth, development and total yield of inoculated chickpea. [19]

IN FOCUS

Additive effects of Phosphorus with inoculation

The interactive effect of seed inoculation (i.e. un-inoculated and inoculated seed) along with various levels of phosphorus (i.e. 0, 60, 90, and 120 kg P_2O_5ha-1) on chickpea growth has been investigated. Chickpea was sown on a sandy clay loam soil, having 0.96% organic matter, 0.06% nitrogen, 6.20 ppm phosphorus, and 118 ppm potassium.

The results revealed that higher 1000-seed weight seed yield and biological yield (256.10 g, 3088.21and 7496.99 kg ha-1, respectively) were obtained with seed inoculation and 90 kg P_2O_5 ha-1 application (Inoc.1×P3). Correspondingly, the lowest seed yield, biological yield, and 1000-seed weight were obtained with un-inoculation and zero applied phosphate (Inoc-0×P1). [20]

3.2 Seed treatments

It is recommended that whenever possible seed should be obtained from a source where the crop was free from Ascochyta blight and Botrytis grey mould.

All seed, regardless of source, should be treated with a registered thiram-based fungicide seed dressing prior to sowing (Table 5).

These seed treatments will help to minimise the levels of seed-borne Ascochyta and Botrytis grey mould. Research has shown that thiram plus thiabendazole products (e.g. P-Pickle-T®) and thiram only products (e.g. Thiraflo®) are equally effective against Ascochyta and Botrytis.

A fungicidal seed dressing to suppress early development of Ascochyta blight is essential. Use thiram or thiabendazole and thiram combined, which is also effective against Botrytis grey mould. There are no known fungicide seed dressings or treatments to control sclerotinia, although grading may assist by physically reducing the number of small sclerotes (fungal fruiting bodies) in the seed sample.

Kabuli chickpeas may show a response to the application of fungicide seed dressings even in the absence of known fungal diseases. This is because Kabulis have a

19 Namvar, A., Sharif, R. S., & Khandan, T. (2011). Growth analysis and yield of chickpea (*Cicer arietinum* L.) in relation to organic and inorganic nitrogen fertilisation. Ekologija, 57(3).

20 Ali, H., Khan, M. A., & Randhawa, S. A. (2004). Interactive effect of seed inoculation and phosphorus application on growth and yield of chickpea (*Cicer arietinum* L.). International journal of Agriculture and Biology, 6(1), 110-112.

thinner seed coat than Desi types and a lower content of phenolic compounds, which help protect the seed against fungal attack. [21]

Table 5: *Seed dressings registered for the control of seed-borne Ascochyta blight and Botrytis grey mould.*

Active ingredient	Example of trade name	Rate (per 100 kg seed)
thiram (600 g/L)	Thiraflo	200 mL
thiram (800 g/kg)	Thiragranz	150 g
thiram + thiabendazole (360 + 200 g/L)	P-Pickel T	200 mL

Refer to the current product label for complete 'Direction for use' prior to application.

MORE INFORMATION

Inoculating pulses

All chickpea seed should be inoculated with Group N inoculant. Fungicide seed dressings can reduce the longevity of this nitrogen fixing bacteria. But if contact is kept to a minimum duration, satisfactory nodulation will be obtained in most cases.

Fungicide seed dressings can be applied at any convenient time leading up to sowing. However, inoculation of seed needs to occur as close as possible to the time of sowing.

All inoculated seed should be sown into moist soil within 12 hours of treatment (or as per the inoculant's label directions), and the sooner the better. If inoculated seed is not sown within 12 hours, re-inoculate before sowing. [22]

Seed treatment is very effective against seed rot, permitting early seeding of Kabuli types to help offset the later maturity of currently available Kabuli chickpea varieties.

If the seed is treated with fungicide, it should be planted immediately after inoculation, as seed treatments can be toxic to the inoculant. The longer the inoculant is in contact with the seed treatment, the less effective it will be. [23]

3.3 Time of sowing (yield losses due to delay; frost risk timing)

Determining the optimum sowing date for pulses is complex, being strongly influenced by factors including environment, management system, cultivar, and grower attitude to risk. The sowing window for most crops in southern Australia is between late April and July. Chickpeas are sown in the middle of this window, and field peas are traditionally sown last.

WUE is commonly in the range of 8–12 kg grain/ha/mm for sowings made during the preferred sowing window. This drops away to 4–6 kg/ha/mm for very late or very early sowings.

Sowing prior to the recommended sowing window tends to result in greater vegetation and crops suffer from:

- poor early pod set because of low temperatures (<15°C) at flowering commencement
- higher risk of Botrytis grey mould at flowering–podding
- greater pre-disposition to lodging
- increased frost risk at early podding
- high water use prior to effective flowering and the earlier onset of moisture stress during podding

21 Pulses Australia. Chickpea Production: Southern and Western region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

22 L Jenkins, K Moore, G Cumming. Pulse Australia. Chickpea: High Quality seed. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/high-quality-seed

23 Pulses Australia. Chickpea Production: Southern and Western region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

- increased risk of Ascochyta blight.

Late-planted crops are more likely to suffer from:

- high temperatures and moisture stress during podding
- greater native budworm pressure
- shorter plants, which are more difficult to harvest.

To achieve maximum yields, critical management factors such as weed control and seedbed preparation must be planned to allow crops to be sown as close as possible to the 'ideal sowing dates'.

Ideal sowing dates should ensure that all chickpea crops:

- finish flowering before they are subjected to periods of heat stress, generally when maximum day temperatures over a week average 30°C or more; and
- flower over an extended period to encourage a better pod set and produce sufficient growth to set and fill an adequate number of pods.

Sowing must not be too early, otherwise:

- flowering may occur during a frost period;
- growth may be excessive, resulting in the crop lodging while dramatically increasing the likelihood of fungal disease problems in the medium–high rainfall districts; and
- conditions at seeding time may not be suitable for controlling broadleaved weeds with recommended herbicides, resulting in weedy crops.

This means that there can be a significant difference between the optimum sowing time for maximum potential yields and the ideal sowing time for reducing yield loss factors. The ideal seeding time for pulses depends largely on where the crops are being grown. Key factors include rainfall and the date of risk periods such as frost and critical heat stress. Soil type and fertility can also influence crop growth. With all pulses, it is essential to have adequate soil moisture at seeding time.

In some areas, the ideal sowing date will be a compromise. Optimum yields achieved by early sowing may have to be sacrificed, with sowing being delayed until risk factors have been reduced to an acceptable level (Figure 5). [24]

Time of sowing has been identified as a major factor affecting chickpea yield and disease incidence. [25] The key to planting chickpeas is to be mindful that the crop is susceptible to stress during flowering. Selecting a planting date that will limit this stress is a practical way to give the crop the best chance of achieving its potential yield.

The later a crop is planted, the shorter the potential season for growth and development, especially if the season has a hot dry spring. When this occurs, plants have less time to develop canopies and roots, resulting in only partial use of soil water and a yield that is below potential. Reducing the row spacing of late-planted crops and ensuring an adequate plant density is one method to help late-planted crops access all available soil water. [26]

Chickpea shows a marked response to time of sowing. Crops sown 'on time' have an excellent chance of producing very high yields. However, crops sown earlier or later than recommended often suffer reduced yields.

Spring sowing is a preferred option in high rainfall areas (greater than 550 mm). Chickpea crops are best sown from mid-May in the 300–400 mm rainfall zone, and mid-June in the 400–500 mm rainfall zone to maximise yield. However, later sowing has been successful in maximising yield while reducing Ascochyta blight and

24 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

25 Knights EJ, Siddique KHM (2002) Chickpea status and production constraints in Australia. In: Integrated management of botrytis grey mould of chickpea in Bangladesh and Australia. Summary Proceedings of a Project Inception Workshop, Bangladesh Agricultural Research Institute, Joydebpur, Gazipur, Bangladesh. (Eds MA Bakr, KHM Siddique, C Johansen). June 2002, pp 33-41.

26 J Whish, B Cocks (2011) Sowing date and other factors that impact on pod-set and yield in chickpea. GRDC Update Papers 20 April 2011, http://cropit.net/sites/default/files/content/files/Sowing%20date%20impact%20on%20pod%20set_Jeremy%20Whish_GRDC%20Update%202011.pdf

fungicide applications. Early sowing increases the risk of Ascochyta blight. Varieties resistant to Ascochyta blight may be sown at the earlier pre-Ascochyta blight sowing dates. [27]

IN FOCUS

Chickpea and lentil trial – Westmere, south-western Victoria

Take home messages:

- Chickpeas and lentils should be sown in early May to achieve early growth and maximise biomass and yield.
- In 2013 at Westmere, growth was generally slow and lacked vigour.
- The larger Kabuli chickpeas—such as PBA Monarch⁽ᶲ⁾, Almaz⁽ᶲ⁾ and Kalkee—are likely to be the most profitable options for this region.

Small variety trials in lentils and chickpea were sown at Westmere to re-investigate the potential of these crops, now that several new varieties have been developed with improved agronomic characteristics. [28]

Yield increases exhibited by winter-sown chickpea have been ascribed to the longer vegetative growth periods, leading to a larger vegetative structure. This larger vegetative structure intercepts photosynthetically active radiation (PAR) more effectively in spring and supports a proportionally larger reproductive sink with adequate partitioning of dry matter. [29]

One study found that chickpea yield per unit area increases with both earlier sowing and increased supplemental irrigation. However, Water Use Efficiency (WUE) under supplemental irrigation decreases with earlier sowing, due to the relatively large increase that occurs in the amount of evapotranspiration at early sowing dates. [30]

27 Pulse Australia. Chickpeas in South Australia and Victoria. http://www.pulseaus.com.au/storage/app/media/crops/2007_Chickpeas-SA-Vic.pdf

28 Brand J. Online Farm Trails/GRDC - Chickpea and Lentil trial (2013). http://www.farmtrials.com.au/trial/16166

29 Croser, J. S., Clarke, H. J., Siddique, K. H. M., & Khan, T. N. (2003). Low-temperature stress: implications for chickpea (*Cicer arietinum* L.) improvement. Critical Reviews in Plant Sciences, 22(2), 185-219.

30 Oweis, T., Hachum, A., & Pala, M. (2004). Water Use Efficiency of winter-sown chickpea under supplemental irrigation in a Mediterranean environment. Agricultural water management, 66(2), 163-179.

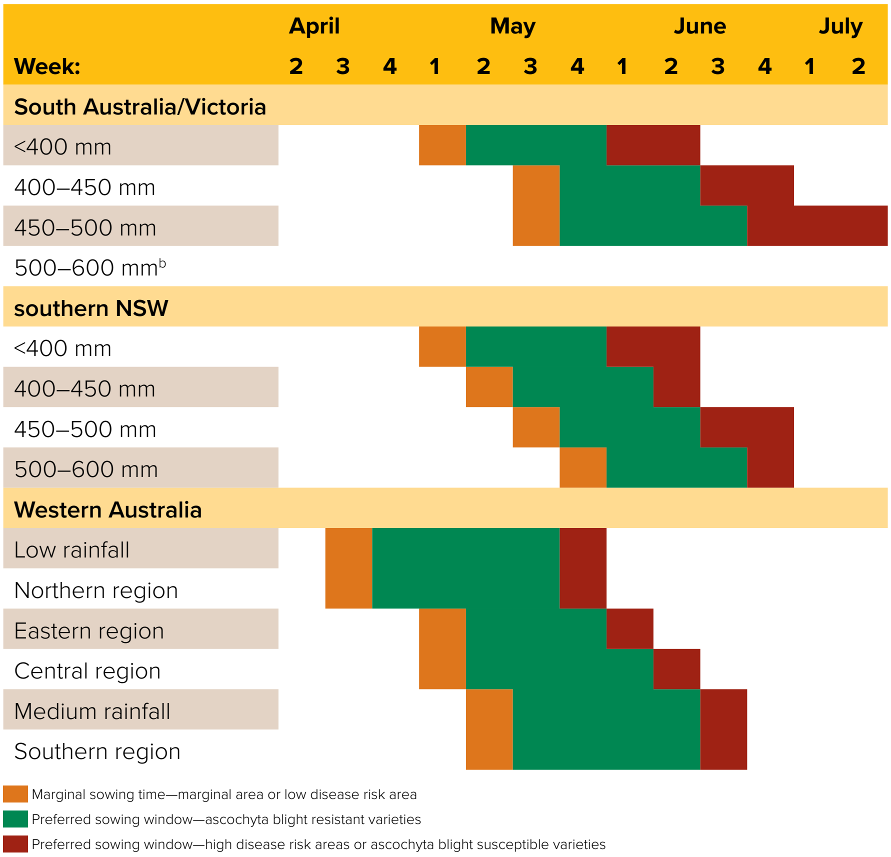


Figure 5: *Preferred chickpea planting times for districts within the Southern growing region. a. Sowing time needs vary with the variety flowering date and maturity rating. b. Preferred sowing time for spring-sown chickpea in south-eastern Australia is August-September.*

Source: Pulse Australia.

IN FOCUS

Improving the reliability of early-sown pulses in south-eastern Australia.

Sowing of pulses in south-eastern Australia generally occurs between early May and late June depending upon crop type and rainfall, but has typically been delayed compared to cereals, due to agronomic and disease limitations. However, recent seasons in this region have experienced dry and hot springs, making earlier sowing of pulse crops essential in many areas to achieve profitable production. Yields of faba beans, field peas, chickpeas, and lentils were compared at two or three sowing dates in small-plot field trials conducted across South Australia and Victoria from 2006 to 2009. First sowing ("early") occurred no more than two weeks after first autumn rains. Subsequent sowings ("mid" and/or "late") were at three-week intervals thereafter, representing more traditional sowing dates. Climate, crop growth, and grain yield data were recorded. [31]

The data illustrates that chickpea can be sown later than the other pulses lentil, field pea and faba bean.

3.3.1 Frost damage

Chickpea seedlings are tolerant of frost. Desi chickpea seed can germinate in soil as cold as 5°C, but seedling vigour is greater if soil temperatures are at least 7°C. Kabuli chickpea seed is more sensitive to cold soils and should not be seeded into excessively wet soil or into soil with temperatures below 12°C at the placement depth.

Damage to vegetative growth:

Damage is more likely to occur where the crop has grown rapidly during a period of warm weather, and is then subjected to freezing temperatures. The visible effect may occur as patches in the field, or on individual plants or branches of plants. Damage is usually more severe where stubble has been retained. Regrowth will generally occur, provided soil moisture levels are adequate.

Damage to flowers and pods:

Freezing temperatures destroy flowers and young developing seeds (Photo 8). Pods at later stage of development are generally more resistant and only suffer from a mottling and/or darkening of the seed coat. Varieties with an extended podding period can compensate for damage better than varieties that tend to pod up over a shorter period provided soil moisture levels are adequate.

Frost is most damaging to yield:

- when it occurs during later flowering–early pod fill
- under dry conditions where moisture limits the plant's ability to re-flower and compensate for frost damage. [32]

31 Lines, M. Improving the reliability of early-sown pulses in south-eastern Australia. Significance, 4(3), 0-2.

32 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

Photo 8: *Frosted chickpea at flowering.*

Source: Pulse Australia.

For more information on frost damage and management in chickpeas, see Section 14: Environmental issues.

3.3.2 Warm season cropping, southern Australia

MORE INFORMATION

Warm season cropping in the southern cropping zone of Australia

Warm season cropping is an emerging opportunity in the Southern region, with early trials underway. The practice involves growing summer crops in the southern cropping belt. These crops include chickpeas, grain sorghum, forage sorghum, corn, safflower, sunflower, millet, and cotton. These crops are sown in spring to mature in late summer/autumn. The idea is to introduce these crops into current rotations as a regular phase of the rotation, not as an opportunity crop that is only sown following a wet harvest. They can be used to diversify rotations, provide breaks to current crop types, improve WUE of current systems and compete with summer weeds. Their agronomy is centred around three fundamental principles: wide row spacings (up to one metre with skips); stubble retention with zero tillage; and paddock preparation to ensure full soil profiles of water at seeding. [33]

Considered the missed opportunity to grow a winter crop in that same season. However, there is increased opportunity to implement agronomic practices, for example to control herbicide resistant weeds before sowing or introduce C4 crops to assist in soil biology.

3.4 Seed rate

Sowing rate affects plant establishment and is an important crop management decision. While yields are relatively stable within the range of 35–50 plants/m^2, higher seeding rates (50 plants/m^2) produce the highest yields in southern and western areas (Table 6). High populations planted on wide rows often result in thin main stems and a higher risk of lodging.

For South Australia, use sowing rates to achieve 30–40 plants/m^2 in the 300–350 mm rainfall zone, and 25–35 plants/m^2 in the 350–500 mm rainfall zones are suggested. For new small Kabuli varieties (e.g. Genesis 090), target a similar plant density to Desi types. [34]

Higher populations are justified for late sowings, while lower populations of around 20 plants/m^2 are often recommended for crops grown on wide row spacing (e.g. 100

33 Wilhelm, N. S. Warm season cropping in the southern cropping zone of Australia. Red, 140(209), 4584.

34 Pulse Australia. Chickpeas in South Australia and Victoria. http://www.pulseaus.com.au/storage/app/media/crops/2007_Chickpeas-SA-Vic.pdf

cm). High populations sown on wide rows often result in thin main stems and a higher risk of lodging. [35]

Table 6: *Seeding rate (kg/ha) required for targeted plants/m² for a range of chickpea varieties at 95% germination and 80% establishment.*

Example variety type		Seed weight (g/100)	Seeding rate (kg/ha):		
			20 plants/m²	25 plants/m²	30 plants/m²
Almaz[⌀]	Large Kabuli	42	111	138	166
Genesis™079	Small Kabuli	26	68	86	103
Genesis™090	Small Kabuli	30	79	99	118
Genesis™114	Large Kabuli	44	116	145	174
Genesis™Kalkee	Larger Kabuli	46	121	151	182
PBA Boundary⌀	Medium Desi	20	53	66	79
PBA Slasher⌀	Medium Desi	20	53	66	79

Source: Pulse Australia.

3.4.1 Calculating seed requirements/sowing rate

Seeding rate for the target plant density can be calculated using germination percentage, 100 seed weight and establishment percentage (Figure 6).

Adjust sowing rates to take account of seed size, germination percentage and estimated establishment conditions.

$$\text{Seeding Rate (kg/ha)} = \frac{\text{100 seed weight\# x Target plant population* x 1000}}{\text{Germination \% x Estimated Establishment \%*}}$$

Example

100 seed weight = 21 grams

Target plant density = 25 plants/m² (i.e. 250,000 plants/ha)

Germination % = 95%

Estimated establishment % = 85%

$$\text{Seeding rate (kg/ha)} = \frac{21 \times 25 \times 1000}{95 \times 80}$$

= 69.08 kg/ha

#100 seed weight in grams from the variety characteristics table.
*Target plant population for your location (seek local advice)

Figure 6: *Seeding rate calculation – Desi chickpea example.*

Source: Pulse Australia.

3.5 Targeted plant population and row spacing

As part of an overall farming system, there is a move towards using row spacing configurations with chickpeas wider than the standard 15 to 25 cm [36].

Trials in southern NSW on row spacings and placement in crop rotations suggest:

1. Sow break crops between standing wheat rows which need to be kept intact
2. Sow the following wheat crop directly over the row of the previous years break crop.

By following these two rules it ensures the following;

35 L Jenkins, K Moore, G Cumming. Pulse Australia. Chickpea: High Quality seed. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/high-quality-seed

36 Felton, W. L., Marcellos, H., & Murison, R. D. (1996, January). The effect of row spacing and seeding rate on chickpea yield in northern New South Wales. In Proceedings of the 8th Australian Agronomy Conference. Queensland, Australia: The Australian Society of Agronomy (pp. 250-253).

(a) ensures four years occur between wheat crops being sown in the same row space.

(b) substantially reduces the incidence of Crown Rot in wheat crops.

(c) Improved germination of break crops, especially canola, not hindered by stubble

(d) Chickpeas will benefit from standing stubble reducing the impact of virus.

(e) Standing wheat stubble gives better protection to break crop seedlings . [37]

Some innovators are sowing in 50 to 100 cm row spacing and using inter-row spraying for weed control. Wider rows require adequate stubble presence to minimize soil evaporative losses and viruses. Weed control must be considered too. Standing stubble and wider rows improve chickpea harvestability and may have advantages in:

- low yielding or lower rainfall situations, or
- when dense canopies would otherwise reduce pod set and lead to BGM.

Fitting the farming system is the important issue. Disadvantages are normally more than offset by the advantages offered by machinery access and zero or minimum tillage systems with stubble retention. [38] The advantage of row-cropping chickpeas outweighs any potential yield reductions as no till weed control methods can be applied and may be the difference between farmers electing to no-till or continue to cultivate their fallow. [39]

Chickpeas are successfully grown using a wide range of planting equipment and row spacings ranging from 18 cm to one metre. Stubble retention, preferably standing stubble, is essential with wide rows.

It is important to consider that pulse row spacing should not be considered in isolation. If choosing wider rows this needs to be undertaken as part of a whole farming system. Stubble presence, minimum soil disturbance inter-row, and good weed control are all vital factors if wide row spcings are used.

3.5.1 Wide rows (50–100 cm) offer:

- Greater ability to plant into heavy stubble cover. Zero tillage systems have shown a consistent 10–15% yield advantage over cultivated systems.
- Precision planters often provide more accurate seed placement, resulting in better establishment and more even plant stands. This often results in more even crop maturity.
- Improved harvestability due to plants being more erect, with a higher pod set as a result of 'within row' plant competition (Photo 9). This is particularly important in low yielding situations.
- In low yield situations, crops planted on wide rows often 'feed in' better over the knife section of the header due to the concentration of growth within the row.
- Reduced input costs through band-spraying of insecticides and defoliants.
- Relatively cheaper weed control using glyphosate through shielded spraying equipment (however, this may have a limited lifespan due to the extent of glyphosate resistance in the Southern cropping region).
- Easier access and 'marking' for ground-spraying pesticides and desiccants in permanent controlled traffic (CT) lanes.
- Better yields under severe moisture stress conditions attributed to the combination of wide-rows and heavy stubble cover than narrow rows configurations (Photo 10).

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37 A Verrell (2013) Row placement strategies in a break crop wheat sequence. GRDC Update Papers 26 Feb 2013, http://cwfs.org.au/wp-content/uploads/2015/11/Row-placement-strategies-in-a-break-crop-wheat-sequence-Grains-Research-Development-Corporation.pdf

38 Pulse Australia. Southern Pulse Bulletin PA 2010 #05 – Chickpea checklist for southern growers. http://www.pulseaus.com.au/storage/app/media/crops/2010_SPB-Chickpea-checklist-south.pdf

39 Felton, W. L., Marcellos, H., & Murison, R. D. (1996, January). The effect of row spacing and seeding rate on chickpea yield in northern New South Wales. In Proceedings of the 8th Australian Agronomy Conference. Queensland, Australia: The Australian Society of Agronomy (pp. 250-253).

- Easier access to the crop when checking for pests such as *Helicoverpa*.
- Improved air circulation in the crop, which lowers humidity levels and can reduce the severity of foliar fungal diseases.
- Inter-row cultivation and 'directed' herbicide sprays, e.g. Broadstrike®.

Photo 9: *Wide row pulses held erect on standing stubble for harvest.*

Source: Pulse Australia.

Photo 10: *Chickpeas planted 1 June on a full moisture profile at 65 kg/ha on 800 mm row spacing and showing good growth despite cold conditions.*

Source: Farmnet.

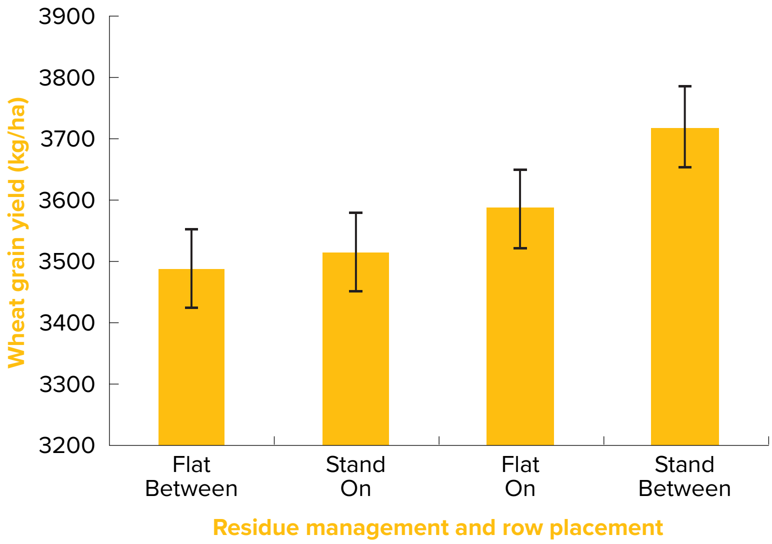

Figure 7: *Effect of row placement (relative to the 2012 wheat crop) and stubble management in the 2013 chickpea crop on grain yield (kg/ha) in the 2014 wheat crop.*

Source: NSW DPI

MORE INFORMATION

Wide rows and stubble retention

3.5.2 Narrow rows (15–40 cm) offer:

- Potential yield advantage at yields levels above 1.5 t/ha. Any yield advantage is often negated, however, by the inability to maintain a zero-till system when planting on narrow row spacings.
- Relatively fewer lodging problems in high yield situations.
- Suits conventional wheat planting equipment. [40]

IN FOCUS

Yield response of Kabuli and Desi chickpea (*Cicer arietinum* L.) genotypes to row spacing in southern Australia

A field experiment was undertaken to investigate the response of Kabuli (three varieties) and Desi (three varieties) chickpea genotypes to row spacing (RS) (18, 36, and 54 cm).

Although Genesis Kabuli genotypes showed lower yield potential (694–1158 kg ha-1) than Desi genotypes (1036–1636 kg ha-1), they were less sensitive to widening row spacing which could be related to their greater branching capacity and appear better suited to wide row cropping systems in southern Australia.

The response of chickpeas to RS was related to the branching habit of the genotype.

40 Pulses Australia. Chickpea Production: Southern and Western region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

MORE INFORMATION

Row spacing of winter crops in broadscale agriculture in southern Australia.

Conclusion

Genesis 090 and 079 Kabuli chickpeas were less sensitive to wider row spacing which could be related to their ability to maintain branching even when crowded by higher neighbour density. Evaluation of germplasm in wide rows should enable selection of cultivars better suited for wide row cropping systems in southern Australia. [41]

3.6 Row placement

A break crop (pulse or oilseed) following a wheat crop should be sown between the standing stubble rows. In the next year, the wheat crop should be sown directly over the previous season's break crop row. Then in the next year of the rotation, the break crop should be shifted back and be sown between the standing wheat rows. Finally, in the fifth year, the wheat crop again should be sown directly over the previous year's break crop row.

There are two simple rules that need to be followed:

1. Sow break crops between standing wheat rows, which need to be kept intact.
2. Sow the following wheat crop directly over the row of the previous year's break crop.

Following these two rules will ensure the following:

- that four years elapse between wheat crops being sown in the same row space
- substantial reduction in the incidence of crown rot in wheat crops
- improved germination of break crops, especially canola, not hindered by stubble
- benefit to chickpeas from standing stubble, reducing the impact of virus infections
- better protection to break-crop seedlings from standing wheat stubble. [42]

3.7 Sowing depth

Depth of sowing is an important agronomic practice affecting the emergence and establishment of crops, especially with early sowing under dryland conditions when temperatures and soil evaporation rates are high. Chickpeas have hypogeal emergence where their cotyledons remain where the seed is sown while only the shoot emerges from the soil surface.

Chickpea seed is best sown into friable soil, with direct drilling often possible following a cereal crop. Good depth and adequate seed-to-soil contact is required and the large seed size of chickpeas assists in this regard.

Deeper sowing depths are used when the top soil layer is dry or where greater "depth" protection is needed from residual herbicides used on the soil surface. However, deeper sowing can result in greater soil disturbance and delayed crop emergence although it helps to reduce lodging of the crop. [43]

Sow chickpeas 5–7 cm deep into good moisture. The seedlings are robust, provided high quality seed is used. The agronomic advantages of sowing at 5–7 cm include:

- reduced risk of damage from pre-emergent residual herbicides such as simazine, Balance etc.

41 Kleemann, S., & Gill, G. Yield response of kabuli and desi chickpea (*Cicer arietinum* L.) genotypes to row spacing in southern Australia. Genesis, 79, 0-10.

42 A Verrell (2013) Row placement strategies in a break crop wheat sequence. GRDC Update Papers 26 Feb 2013, http://cwfs.org.au/wp-content/uploads/2015/11/Row-placement-strategies-in-a-break-crop-wheat-sequence-Grains-Research-Development-Corporation.pdf

43 http://www.regional.org.au/au/asa/2012/crop-production/8197_haighb.htm

- improved early formation of lateral roots in the top soil
- enhanced inoculum survival in moist soil
- a significant proportion of Ascochyta-infected seed is eliminated due to high mortality of diseased seed.

Avoid sowing deeper than 5 cm on soils prone to surface sealing and crusting.

Press-wheels can improve establishment, although heavy pressures should be avoided. V-shaped press-wheels will leave a furrow down the planting line that can lead to a concentration of residual herbicides in the furrow after rainfall and subsequent crop damage. [44]

One recommendation is that seed should be sown at a depth of 25 to 80 mm. On clay soils, a minimum depth of 50 mm is suggested. This sowing depth can also assist in preventing Ascochyta-infected seed from emerging. On light textured soils, shallow sowing can lead to pre-emergent herbicide damage. On heavy textured soils, surface sealing and deeper sowing delays emergence, reduces plant establishment and early plant vigour. Chickpeas are slow to emerge. Later sowing can take four weeks or more to emerge with cold conditions. If simazine or metribuzin is used for weed control in a wet winter, leaching can cause root damage. On lighter soils, leaching into the root zone will be more of a problem. [45]

It is generally recommended that chickpea seed should be sown 5–7 cm deep into moist soil. The preferred depth when using Balance® or simazine herbicides is 7 cm.

Sowing poor quality seed too deeply, into cold and/or wet soils, hard setting (crusting) soils and with some PSPE herbicides can reduce the ability of the germinating seedling to quickly reach the soil surface, which increases the seedling's susceptibility to both soil and seed-borne pathogens, and soil-borne insect pests.

Moisture seeking strategies (i.e. planting at a depth of 10–15 cm below the soil surface) should be avoided for all weather damaged or low vigour seed. [46]

One study showed that lodging increased with shallower (5 cm vs 10 cm) sowing depth on the grey clay soil. [47]

IN FOCUS

Sowing Depth for Chickpea (*Cicer arietinum* L.), Faba Bean (*Vicia faba* L.) and Lentil (*Lens culinaris Medik*) in a Mediterranean-type Environment of southern Australia

Pulses such as chickpeas, faba beans, and lentils have hypogeal emergence and their cotyledons remain where the seed is sown, while only the shoot emerges from the soil surface. The effect of three sowing depths (2.5, 5, and 10 cm) on the growth and yield of these pulses was studied at three locations across three seasons in the cropping regions with a Mediterranean-type environment. There was no effect of sowing depth on crop phenology, nodulation, or dry matter production for any species. Mean seed yields across sites ranged from 810 to 2073 kg ha–1 for chickpeas, 817–3381 kg ha–1 for faba beans, and 1173–2024 kg ha–1 for lentils. In general, deep sowing did not reduce seed yields, and in some instances, seed yield was greater at the deeper sowings for chickpeas and faba beans. We conclude that the optimum sowing depth for chickpeas and faba beans is 5–8 cm, and for lentils 4–6 cm. Sowing at depth may

44 Pulses Australia. Chickpea Production: Southern and Western region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

45 Pulse Australia. Chickpeas in South Australia and Victoria. http://www.pulseaus.com.au/storage/app/media/crops/2007_Chickpeas-SA-Vic.pdf

46 L Jenkins, K Moore, G Cumming. Pulse Australia. Chickpea: High Quality seed. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/high-quality-seed

47 http://www.regional.org.au/au/asa/2012/crop-production/8197_haighb.htm

also improve crop establishment where moisture from summer and autumn rainfall is stored in the subsoil below 5 cm, by reducing damage from herbicides applied immediately before or after sowing, and by improving the survival of Rhizobium inoculated on the seed due to more favourable soil conditions at depth. [48]

3.7.1 Deep seeding strategies

Deep planting is dependent on soil type, and is not commonly practiced in the Southern cropping region. However, deep planting has proven to be an extremely valuable risk management tool in many seasons in northern Australia. It has allowed chickpea to be planted in situations where winter crop planting rains have either been very late or have failed to eventuate altogether.

In these years, a large proportion of the northern Australian chickpea crop is planted using deep-planting techniques. Crops are commonly planted at depths of 10–20 cm and this has often been the difference between achieving a reasonably profitable crop or no crop at all.

Colder soil temperatures, wetter soils and slower emergence times needs to be considered when deep planting chickpeas in southern Australia. A 'sow deep, cover shallow approach' is needed, being wary of potential damage from herbicide wash into the furrow after rain.

In arid regions, chickpeas are sown deep as surface moisture is often inadequate to allow adequate crop establishment. Deep-planting is not only an extremely valuable tool under drought conditions, but can also offer major advantages in most years. It allows growers to plant chickpeas at the optimum time for their district, regardless of highly variable rainfall events. This maximises crop WUE, grain yield, crop height, and profitability.

Critical issues that need to be addressed include:

- seed quality and vigour
- planter configuration and operation
- filling in the seed trench (furrow)
- harvestability of the crop

Deep planting checklist

- **Plan ahead if you are considering deep-planting of chickpeas.** Growers can use this technique in most years to ensure that they plant at the optimum time for chickpeas in their district rather than relying on highly variable rainfall events. This is preferable to using deep planting as a last resort, salvage operation after planting rains fail to eventuate and the optimum-planting window has already passed. **The key to deep-planting is to make the decision early and sow on time.**
- **Exercise caution** when deep-planting on hard setting or crust-prone soils.
- **Decide on the best combination of sowing point, press-wheel, and operational speed for your planter and soil type.** Be prepared to alter this combination depending on soil conditions at the time of planting. Speed is critical as it can have a major impact on depth control, as well as the amount of soil coverage over the seed.
- **Ensure you have high quality planting seed.** The deeper you plant, the greater the importance of using high quality planting seed. Check your germination percentage and seed counts (seeds/kg) and adjust seeding rates accordingly.

48 Siddique, K. H. M., & Loss, S. P. (1999). Studies on sowing depth for chickpea (*Cicer arietinum* L.), faba bean (Vicia faba L.) and lentil (Lens culinaris Medik) in a Mediterranean-type environment of South-western Australia. Journal of Agronomy and Crop Science, 182(2), 105-112.

Only use the highest quality seed when attempting deep planting. There are two additional seed tests that can be used to better determine seed quality.

The Accelerated Ageing (AA) Test. This test is normally undertaken after harvest or well before planting and gives an indication of the seed vigour at planting time providing storage conditions are good. The value of this test is that seed showing poor vigour can be identified early and alternative actions can be taken. This test is highly recommended for seed that is likely to be deep-sown. A germination test should also be done at the same time. If the results from the AA test are similar to the germination test, then the seed has good vigour. If there is a significant difference between the two tests then advice on the interpretation of the test should be sought.

The Vigour or Soil Germination Test. This test is recommended prior to planting and gives a guide to seed vigour in soil conditions at that time. The guidelines for interpreting the results of this test are the same as for the AA test above.

- **Increased weed pressure.** When deep planting under dry conditions, the first general winter rain will now fall in-crop and winter weeds will germinate on this in-crop rainfall.
  This places a lot more pressure on broadleaf and grass weed control as growers can no longer rely on a non-selective knockdown spray prior to planting to tidy up winter weeds.
  Growers need to ensure that they have an appropriate weed strategy mapped out before planting.
- **Use fungicide treated seed.** As a precaution against the seed transmission of Ascochyta blight.
- **Spray out fallow weeds prior to planting.** These can be difficult to control if moisture-stressed and covered in dust (because of the dry conditions). Adjust herbicide rates and water volumes accordingly.
- **If you are using residual herbicides such as Balance® or simazine you will need to fill in the furrow (seed trench) prior to applying the herbicide.** If you cannot fill in the trench completely, then you should at least ensure you have 8–10 cm of soil coverage above the seed. Both these measures will ensure that the risk of herbicide damage after rain is minimised.
- **Avoid deep-planting into compacted wheel-tracks** as it usually results in variable depth control and poor seed coverage. Both are major contributors to patchy, uneven plant stands. Adopt the use of controlled-traffic systems wherever possible.
- **Decide on a planting depth that will ensure that all seeds are planted into moisture.** Thoroughly inspect seedbed moisture levels across paddocks and different soil types and ensure you plant into moisture. Experience indicates that you are better to err on the “deeper side” rather than plant “too shallow” into marginal moisture.
- **Ensure that the planter can maintain uniform depth control across the full width of the machine under normal operational speeds.** Poor or variable depth control will result in gappy, uneven plant stands.
- **Harvestability is a major issue.** Deep-planted crops can experience adverse, dry conditions where crop height and harvestability are significant problems.

Deep planting method

The technique referred to as 'moisture seeking' has been used to a limited extent for over 20 years to plant cereals into stored fallow moisture without a planting rain. The practice usually requires the deliberate formation of a furrow or trench above the seed row because wheat and barley have relatively short coleoptiles, which limit the depth of soil they can successfully emerge through to approximately 8 cm.

This practice is referred to as 'deep-furrow planting' because the furrows are deliberately left intact at the completion of the planting operation.

Sweeps or 'shovels' may need to be mounted on the sowing tine assemblies to help shift dry soil out of the furrow.

MORE INFORMATION

Chickpea: Deep Seeding strategies.

Sowing strategies to improve productivity of crops in low rainfall sandy soils

This technique of 'deep-furrow planting' is far less suited to crops such as chickpea for two very good reasons:

- The short stature of the chickpea crop and the need to set the header front as close to the ground as possible
- The reliance on using pre-emergent, residual herbicides for broadleaf weed control. These herbicides can concentrate in the furrow after rain and cause considerable crop damage.

The more appropriate technique for chickpea is 'deep-planting' where growers fill in the furrow and level the soil surface after planting and rely on the chickpea plants' ability to emerge from depth to achieve crop establishment.

Levelling the soil surface considerably reduces the risk of herbicide residue damage and minimises harvest difficulties. [49]

3.8 Sowing equipment

There are few problems when sowing Desi and most Kabuli chickpeas with conventional seeding equipment, but occasionally cracking of seed may occur with the larger seeded Kabuli types. [50]

Ensure that the seed handling equipment and seeder is not too aggressive on the seed (e.g. use shifters instead of augers and avoid high blower speeds in air seeders). [51]

Success with pulses may depend on the type of sowing equipment used. The large size of pulses can make sowing with conventional seeders extremely frustrating. If your seeder is not suitable for sowing a particular pulse (usually larger seeded types) in standard form there are several options available. The machine may be adapted by minor modifications such as:

- modifying the metering mechanism using manufacturer supplied optional parts
- modifying seed tubes to reduce blockages, particularly on older machines
- modifying or replacing dividing heads on airseeders.

Most pulse seeding problems are related to seed metering and the transfer from seed meter to soil. These problems are caused by the large size of some pulses and the high seeding rates generally used.

Kabuli chickpeas can be sown with a standard airseeder or conventional combine but care should be taken, as seeds tend to bridge over the outlets, causing very uneven sowing. This difficulty can be eliminated by filling the box to only a third or a half capacity or by fitting an agitator.

A recent set of trials in southern NSW found that chickpea grain yield increased when sown with a disc opener (by 6%), while a coulter-tyne-press wheel out-yielded the disc opener (by 6.3%). [52]

See Section 3.5.

3.8.1 Seeding equipment – Southern region

Southern Farming Systems (SFS) has conducted extensive work on seeding equipment performance in relation to stubble retention. These include Seeder Demonstration Day 2015, the Streatham disc versus tyne trial, stubble management comparisons and a stubble management for disc seeders trial at Streatham. Key information thus far from this work is:

49 Pulse Australia. Chickpea: Deep seeding strategies. Australian Pulse bulletin.

50 Pulses Australia. Chickpea Production: Southern and Western region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

51 Pulse Australia. Southern Pulse Bulletin PA 2010 #05 – Chickpea checklist for southern growers. http://www.pulseaus.com.au/storage/app/media/crops/2010_SPB-Chickpea-checklist-south.pdf

52 Verrell, A. (2016). GRDC Update Papers. Integrated management of crown rot in a chickpea – wheat sequence. https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/02/Integrated-management-of-crown-rot-in-a-chickpea-wheat-sequence

- Real time kinematic (RTK) guidance is a critical component to inter-row sowing.
- Each seeder has varying capacity to handle retained stubble.
- As a rule, discs handle higher loads than tine and press wheel machines.
- Wider tine spacing across and along the bar will improve stubble handling.
- Changing the angle of sowing direction slightly can minimise blockages.
- Guidance auto steer on seeder bars will improve inter-row sowing.
- Tines and discs have varying degrees of soil throw and crop safety for pre-emergent herbicides.
- Isolation of fertiliser from seed will limit seed burn. [53]

3.8.2 Rolling

Rolling after sowing is desirable on most soils to provide a level surface for herbicide application, and for easier harvesting free of stones and clods. This reduces herbicide washing into seed furrows, harvester wear, and soil contamination in the seed sample. If the soil is prone to hard setting, crusting or erosion on sandy or sloping country, rolling should be delayed until the crop is 15 to 25 cm high. Avoid rolling chickpeas when plants are just starting to emerge as emerging shoots will be broken off. If rolling chickpeas post-emergent, choose a time when the plants are limp such as in the afternoon to reduce the amount of damage. Rubber-tyred rollers are preferable, but steel rollers can be used. Avoid rolling either two weeks before or after applying a post-emergent herbicide. [54]

3.8.3 Combine seeders

Combines with fluted roller feeds such as Chamberlain, Connor Shea, old Napier, and some Massey combines have few problems feeding seed of <15 mm down to the metering chamber.

Combines with peg roller and seed wheel feeds (newer Napier, Shearer, Chamberlain-John Deere) will seed grains up to the size of Kabuli chickpeas without problems, provided adequate clearances are used around the rollers. Smaller faba beans can be metered with the more aggressive seed wheel system, but peg rollers are best replaced with 'rubber stars' for larger faba beans. Broad beans can be metered through the rubber stars, but how efficiently combines sow these seeds is still in question. Combines with internal force-feed seed meters perform well on small seeds but cannot sow seed >9 mm because of bridging at the throat leading to the seed meter. The restricted internal clearance in this type of design can damage larger seeds.

3.8.4 Airseeders

Airseeders that use peg-roller metering systems (Napier, Shearer) will handle grain up to the size of smaller faba beans without problems because of the banked metering arrangement. The optional rubber star roller will be necessary for larger seeds such as broad beans. Airseeders using metering belt systems (Fusion, Alfarm, Chamberlain-John Deere, New Holland) can meter large seed at high rates with few problems. On some airseeders, the dividing heads may have to be modified because there is too little room in the secondary distributor heads to allow seeds to flow smoothly. Photo 11 shows a standard secondary distributor head (on the left) and a conversion to suit Connor Shea airseeders. The conversion head increased the bore from 23 mm to 41 mm. Four larger hoses replace the original eight, and row spacings are increased from 150 mm to 300 mm. This conversion allows large seeds such as Kabuli chickpea or beans to be sown easily. Consult the dealer about possible modifications. Significant levels of seed damage can be caused in

53 Breust P, Vague A. GRDC Update Papers (2016). Profitable stubble retention systems for the high rainfall zone. https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/02/Profitable-stubble-retention-systems-for-the-high-rainfall-zone

54 Pulse Australia. Chickpeas in South Australia and Victoria. http://www.pulseaus.com.au/storage/app/media/crops/2007_Chickpeas-SA-Vic.pdf

airseeders by excessive air pressure, so be careful to use only enough air to ensure reliable operation.

Photo 11: *Conversion heads, such as this one for a Connor-Shea airseeder, allow large seeds such as broad beans, faba beans, and Kabuli chickpeas to be sown with ease.*

Source: Grain Legume Handbook, 2008.

3.8.5 Seeder and tine comparisons

In the establishment of all crops, especially pulses, there are several key functional or mechanical issues with respect to seeding equipment, which should:

- Have an adequate seeding mechanism to handle the pulse seed without damaging it, especially when larger seeded types are being sown.
- Have adequate sizes of seed and fertiliser tubes and boots to prevent seed blockages and bridging during sowing.
- Sow into stubbles and residues, without blockages.
- Have sufficient down-pressure to penetrate the soil, sow at the desirable depth and place all seeds at a uniform depth.
- Cover the seeds to ensure good seed-to-soil contact and high moisture vapour, which will promote rapid germination.
- Compact the soil as required, by press-wheels or closers (Photo 12) (otherwise, a prickle chain or roller is required afterwards for many pulses).
- Disturb the soil to the extent required, which means none in no-till with disc sowing. It may also mean having sufficient soil throw to incorporate herbicides like trifluralin. This can be achieved by using either aggressive discs or narrow point set-ups in no-till, or full disturbance in more conventional or direct-drill systems.

Inability to get adequate plant establishment is one of the bigger problems faced by pulse growers. This can lead to a multitude of problems later. Many different seeding mechanisms or openers are now available to pulse growers. Narrow points are widely used in minimum or no-till systems, but many different points can be used. Likewise, with disc seeders, many different types are now available, and they differ greatly in their soil disturbance and soil throw, as well as their ability to handle trash and sticky conditions.

Photo 12: *One of several seeding mechanisms for uniform sowing depth using the press wheel for depth control.*

A comparison of the key functions that are critical for seed drills and no-till is shown in Table 7.

In interpreting the functions listed in Table 7 it should be noted that:

- With tines, the slot created is different depending on the type of tine used. Some create a vertical slot, others a 'V', while the inverted 'T' (or 'baker boot') leaves a slot with a narrow entrance and wider trench underneath (Photo 13). These tines do perform differently in some functions in Table 7.
- Residues need to be handled in all conditions, not just when dry.
- 'Hairpins' (stubble is pressed into the slot) needs to be avoided by not creating them or by placing seeds away from them. Note that tines rarely make hairpins.
- Vertical slots are hard to self-close.
- Ability for openers to follow ground-surface variation is critical for uniform depth of sowing (Photo 14).
- Springs cannot apply consistent down force on openers throughout a range of soil conditions.
- Banding of fertiliser away from the seed is important for crop establishment, particularly when high rates or high-analysis products are applied and the seed is in a narrow opening slot.
- Tines handle stones, but bring them up, hence requiring rolling to press them back again.

Table 7: *Comparison scores (rating basis: 1, poor; 5, excellent) of no-till openers by function (after Baker 2010)*

	Narrow point	Wide point	Sweep	Double disc	Single disc	Slanted disc	Combined winged tine & disc[B]
Ability to mechanically handle heavy residues without blockage	2	1	1	4	4	4	5
Leave 70%+ of original residue in place after drill has passed	3	2	2	5	4	4	5
Trap moisture vapour in the seeding slot in dry soils using residues as slot cover	3	2	3	1	2	4	5
Avoid placing seeds in 'hairpins'	5	5	5	1	2	2	5

	Narrow point	Wide point	Sweep	Double disc	Single disc	Slanted disc	Combined winged tine & disc[B]
Maximise in-slot aeration in wet soils[A]	3	4	3	1	3	3	5
Avoid in-slot soil compaction or smearing in wet soils[A]	1	1	3	1	5	5	5
Maximise soil-seed contact, even in greasy or 'plastic' conditions	4	3	4	3	3	4	5
Self-close the seeding slots	2	1	3	2	3	4	5
Mitigate slot shrinkage when soils dry out after sowing[A]	3	5	5	1	2	4	5
Individual openers faithfully follow ground surface variations	2	1	2	2	4	2	5
Individual openers have a larger than normal range of vertical travel	2	1	1	2	2	1	5
Maintain consistent down force on individual openers	3	1	1	2	3	3	5
Openers seed accurately at shallow depths[A]	2	1	1	2	2	1	5
Opener down force auto-adjusts to changing soil hardness	1	1	1	1	1	1	5
Simultaneously band fertiliser with, but separate from, the seed	5	5	5	1	2	3	5
Ensure that fertiliser banding is effective with high analysis fertilisers	5	5	51	1	1	2	5
Be able to handle sticky soils[A]	5	5	4	1	3	3	2
Be able to handle stony soils[A]	4	3	1	4	4	2	4
Avoid bringing stones to the surface[A]	1	1	1	5	5	3	5
Functionality unaffected by hillsides[A]	5	5	4	5	2	1	5
Minimal adjustments required when moving between soil conditions	3	3	3	4	1	1	5
Ability to maintain most critical functions at higher speeds of sowing	3	1	1	4	3	3	5
Wear components are self-adjusting	5	5	5	3	2	2	5
Design life of machine matches that of the tractors that pull it	4	4	4	2	2	2	5
Low wear rate of soil-engaging components	5	4	4	2	3	3	3
Wear components, including bearings, are cheap and easily replaced	5	5	4	2	2	2	4
Requires minimal draft from tractor	4	3	2	5	4	3	3
Proven, positive impact on crop yield	3	2	2	1	3	4	5
Total score (maximum = 140)	**93**	**80**	**80**	**68**	**77**	**76**	**131**
Rating score as % of maximum possible	**66**	**57**	**57**	**49**	**55**	**54**	**94**

Note that this table is a broad guide only. Scores given in this table are subjective and may vary with individual openers, etc. You may wish to use your own scores for each function and not count those not relevant to your situation.

In Table 7, neither pure-disc nor pure-tine openers rate highly over all functions using this scoring. Disc openers rated lowest (49–55%), and of the tines (57–66%), narrow points were the best (66%). The combination of winged tine and disc, known as the Bio Blade or Cross Slot™, had the highest score (94%). It allegedly combines the best attributes of pure disc openers with the best attributes of pure tine openers, and adds some unique features of its own. Its weaknesses were its lesser ability to handle 'sticky' soils, its horsepower requirement, and its wear rate of soil-engaging components.

Use Table 7 as a guide only to help select your own openers to suit your conditions and circumstances.

Photo 13: *A Primary Precision Seeder fitted with hydraulic breakout for consistent penetration. It is also fitted with narrow points that form an 'inverted T' slot and is capable of deep or side placement of fertiliser.*

Source: NSW DPI

Photo 14: *The DBS system parallelogram for uniform seeding depth and deep placement of seed or fertiliser.*

The seeding mechanism of the seeder must be able to handle pulses, which are larger seeded than cereals and oilseeds. Hoses, distributor heads, and boots must also be able to handle pulses without blockages or bridging. This is especially true for larger seeded types such as Kabuli chickpeas or faba and broad beans (Photo 15).

Photo 15: *Bio Blade or Cross slot™ disc opener with opening disc and seeding tine, followed by paired press wheels. Note that the seed and fertiliser tube has sharp bends and may not be wide enough to avoid blockages when larger seeded pulses like faba or broad beans are being sown.*

Table 9 does not list as a function deep working to assist in rhizoctonia control. This was a weakness of early disc drills compared with narrow points with deep openers. Many newer discs are addressing this issue, including using opening coulters and rippled discs (Photo 16). [55]

Photo 16: *A Case IH SDX-40 single-disc drill.*

55 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

Plant growth and physiology

Key messages

- Under optimum moisture and temperature conditions, chickpea seeds imbibe water quickly and germinate within a few days, provided temperatures are >0°C.
- Emergence occurs 7–30 days after sowing, depending on soil moisture and temperature conditions and depth of sowing.
- Flowering is invariably delayed under low temperatures but more branching occurs.
- Chickpea in its reproductive stage is sensitive to heat stress (35/20°C or higher as day/night temperatures) with consequent substantial loss of potential yields at high temperatures. In Australia, drought stress often accompanies high temperatures in spring, causing the abortion of flowers, immature pods, and developing seeds.
- Chickpea is a photoperiod-sensitive, long-day plant, where flowering is delayed as day length becomes shorter than a base photoperiod (17 hours).
- Starting soil water can have a strong influence on the yield expectation of chickpea as well as the riskiness of production.

Chickpea, being a legume, belongs to the botanical family of Fabaceae, subfamily Faboideae. It is a semi-erect annual with a deep taproot. Worldwide, two main types of chickpea, Desi and Kabuli are cultivated. Kabuli types, grown in temperate regions, are large-seeded and mainly consumed as a whole seed, whereas Desi types, grown in semiarid tropical and subtropical regions, are mainly consumed as split dhal or turned into flour. Chickpea seed contains about 20% protein, 5% fat and 55% carbohydrates.

The phenology of most crops can be described using nine phases:

1. Sowing to germination
2. Germination to emergence
3. A period of vegetative growth after emergence, called the basic vegetative phase (BVP), during which the plant is unresponsive to photoperiod
4. A photoperiod-induced phase (PIP), which ends at floral initiation
5. A flower development phase (FDP), which ends at 50% flowering
6. A lag phase prior to commencement of grain-filling (in chickpea this period can be very long, up to two months in some cases, under cool temperature conditions (<15°C), which inhibit pod set and pod growth)
7. A linear phase of grain filling
8. A period between the end of grain-filling and physiological maturity
9. A harvest-ripe period prior to grain harvest

These stages of development are generally modelled as functions of temperature (phases 1–8) and photoperiod (phase 4).

Chickpeas are a medium-duration crop, usually beginning flowering within 90–110 days of planting, depending on photoperiod and temperature (Figure 1). Chickpea is a photoperiod sensitive, long-day plant, where flowering is delayed as day length becomes shorter than a base photoperiod (17 hours). [1]

1 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

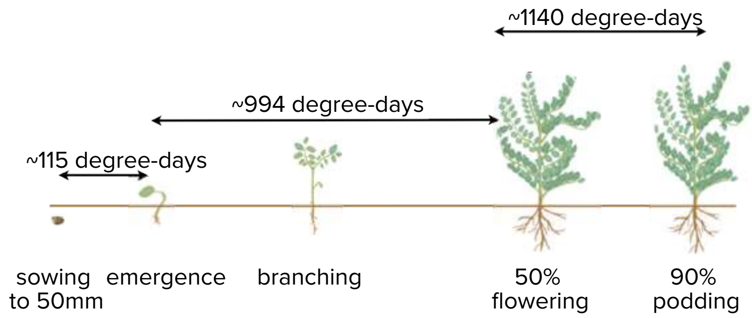

Figure 1: *Key developmental stages of chickpea and their thermal time targets.*

Source: J. Whish, CSIRO

4.1 Germination and emergence issues (e.g. effect on sowing depth, stubble in the paddock, chemical damage)

4.1.1 Germination

Good germination and seedling emergence are important prerequisites for a successful crop and soil and air temperature is one of the key factors affecting seed germination. Under optimum moisture and temperature conditions, chickpea seeds imbibe water quickly and germinate within a few days, provided temperatures are >0°C. Chickpeas will not germinate in soils with temperatures below 0°C. Generally, the longer a germinating seed of a sensitive species is exposed to a chilling temperature, the greater the injury it will sustain. Desi types generally suffer less damage from low temperatures at germination than Kabuli types. Visual symptoms of chilling injury at the seedling stage can include the inhibition of seedling growth, accumulation of anthocyanin pigments, waterlogged appearance with browning of mesocotyls, and the browning and desiccation of coleoptiles and undeveloped leaves. The main effects of chilling range temperatures on the developing seedling are related to membrane injury and include reduced respiration and photosynthesis and loss of turgor, resulting in wilting and cold-induced water stress. Exposure to chilling range temperatures during early growth of established seedlings can exert macroscopic formative effects on leaf shape and size, plant height, root development, and floral initiation. [2]

Chickpea germination is hypogeal, with the cotyledons remaining below the soil surface (Figure 2). This enables it to emerge from sowing as deep as 15 cm. In arid regions, chickpea is sown deep as surface moisture is often inadequate to allow adequate crop establishment (Photo 1). [3]

Note: Chickpea seedlings would not be able to emerge in southern Australia if sown at such depth as in Photo 1 because of low soil temperatures that occur at normal sowing times for chickpea.

2 Croser, J. S., Clarke, H. J., Siddique, K. H. M., & Khan, T. N. (2003). Low-temperature stress: implications for chickpea (*Cicer arietinum* L.) improvement. Critical Reviews in Plant Sciences, 22(2), 185-219.

3 Pulse Australia. Chickpea Production: Southern and Western Region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

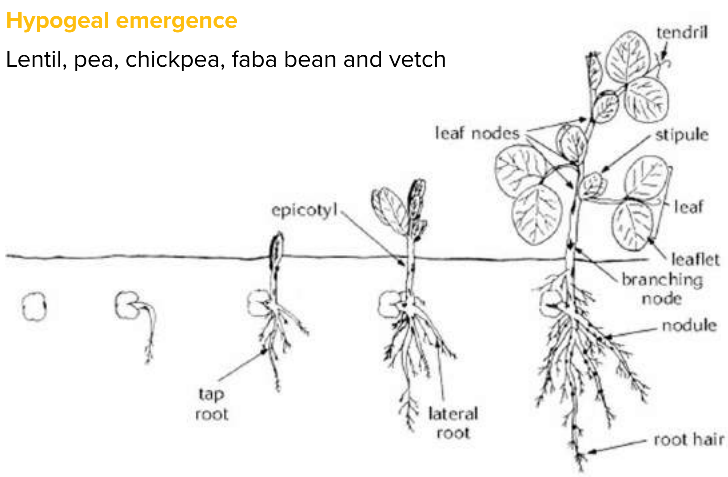

Figure 2: *Hypogeal emergence of chickpea seedlings makes the plant less prone to environmental stress and damage in the early growth stages.*

Source: Pulse Australia.

Photo 1: *A chickpea seedling, sown deep for a dry 2016 start.*

Source: Extension hub - Photo Stephen Gibson.

One of the environmental constraints for the germination is the temperature that negatively affects the seed germination. In fact, the optimum temperature for maximum final germination is between 10 and 15°C. Low temperatures are a major constraint for improving the yield of chickpea in numerous regions of the world.

In one study, all chickpea seeds were able to germinate on a wide thermal range (15–35°C). However, the thermal optimum was about 20–25°C with 80 to 100% of seeds germinated within seven days between 10 and 30°C. [4]

Salinity is one of the major stresses especially in arid and semiarid regions, which severely limits crop production. It impairs seed germination, reduces nodule formation, retards plant development and reduce crop yield. Salinity affects germination and physiology of crops due to osmotic potential which prevents water up take and by toxic effect of ions on embryo viability. [5]

One study found that small seeds germinated and grew more rapidly compared to medium and large seeds under salt stress. The study also found that though there was no effect of NaCl treatments on frequency of germination, there was a drastic decrease in early seedling growth under increased NaCl concentrations. [6]

Altered growth hormone balance during germination is another factor resulting in poor germination and seedling growth under salt stress conditions. Application of growth regulators like gibberellic acid and kinetin have been found to increase germination (32%), root (32%) and shoot (153%) dry mass of seedlings under salt stress.

Salt stress is thought to reduce germination either by making less water available for imbibition or by altering enzymatic activity, growth regulator balance or protein metabolism in germinating seeds. One study has found that pre-soaking seeds for 24 hours in normal ground/tap water (0.8 dS m-1) increased germination by 27% compared to direct sowing in saline conditions. Sowing at 4 cm depth also increased seedling growth under saline soils compared with 2 and 6 cm depths. [7]

4.1.2 Emergence

Emergence occurs 7–30 days after sowing, depending on soil moisture and temperature conditions and depth of sowing. Growth of the shoot (plumule) produces an erect shoot and the first leaves are scales. The first true leaf has two or three pairs of leaflets plus a terminal one. Fully formed leaves with 5–8 pairs of leaflets usually develop after the sixth node.

Photo 2: *Inspecting chickpea plants in the early growth stage.*

Source: GRDC.

4 Sleimi, N., Bankaji, I., Touchan, H., & Corbineau, F. (2013). Effects of temperature and water stresses on germination of some varieties of chickpea (*Cicer arietinum*). African Journal of Biotechnology, 12(17).

5 Haileselasie, T. H., & Teferii, G. (2012). The effect of salinity stress on germination of chickpea (*Cicer arietinum* L.) land race of Tigray. Current Research Journal of Biological Sciences, 4(5), 578-583.

6 Kaya, M., Kaya, G., Kaya, M. D., Atak, M., Saglam, S., Khawar, K. M., & Ciftci, C. Y. (2008). Interaction between seed size and NaCl on germination and early seedling growth of some Turkish cultivars of chickpea (*Cicer arietinum* L.). Journal of Zhejiang University SCIENCE B, 9(5), 371-377.

7 Samineni S. (2010). Physiology, genetics and QTL Mapping of Salt Tolerance in Chickpea.

IN FOCUS

Modelling seedling emergence in chickpea as influenced by temperature and sowing depth

Quantitative information about temperature and sowing depth effects on seedling emergence in chickpeas is scarce.

Six physiological days (equivalent to a thermal time of 94°C days) was required from sowing to emergence at a sowing depth of 5 cm. The physiological days requirement increased by 0.9 days for each centimetre increase in sowing depth.

Based on the results from the field and pot experiments, a seedling emergence model was constructed. This model successfully simulated emergence date (range 4–140 days) in spring, winter and 'dormant' sowing dates across Iran. Using an example for north-west Iran, it was shown how this model could be used to optimise sowing management, including the local 'dormant sowing' practice, whereby the crop is sown prior to winter for early emergence in the following spring. [8]

The node from which the first branch arises on the main stem above the soil is counted as node one. In chickpeas, alternate primary branches usually originate from nodes just above ground level (usually one to eight primary branches on the main stem, depending on growing conditions). A node is counted as developed when 6–15 leaflets have unfolded and flattened out (Photo 3). [9]

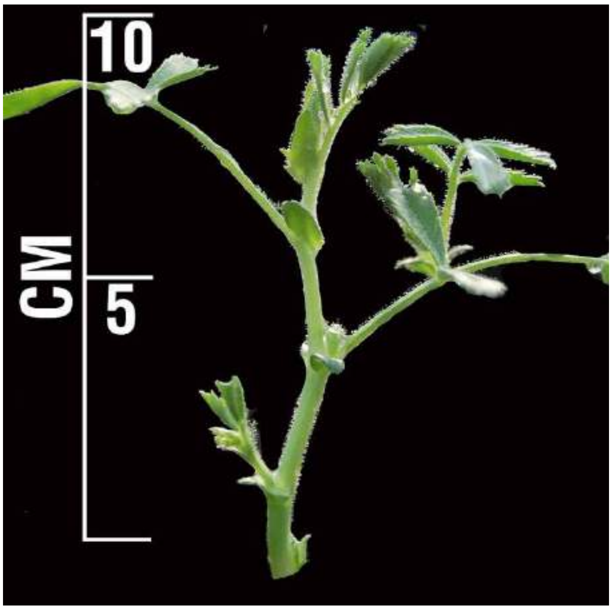

Photo 3: *Chickpea plant with four branches at 8 cm tall.*

Soil water content at sowing is an important determinant of chickpea seed emergence and early growth.

8 Soltani, A., Robertson, M. J., Torabi, B., Yousefi-Daz, M., & Sarparast, R. (2006). Modelling seedling emergence in chickpea as influenced by temperature and sowing depth. Agricultural and Forest Meteorology, 138(1), 156-167.

9 Pulse Australia. Chickpea Production: Southern and Western Region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

IN FOCUS

Effect of Soil Moisture Content on Seedling Emergence and Early Growth of Some Chickpea (*Cicer arietinum* L.) Genotypes.

A controlled glasshouse investigation at day/night temperatures of 22/15 °C was performed (in 2006) to assess the influence of different soil moisture contents (field capacity percentage basis) on emergence, as well as early plant growth in 20 chickpea genotypes.

Significant differences (P < 0.001) regarding plant emergence and early growth were observed among different soil moisture contents (from 100 to 50, then to 25% field capacity) (Figure 3 and Table 1). This brought about a reduction in mean emergence percentage, delayed the first day to emergence and suppressed the early growth in all the chickpea genotypes.

An inverse relationship between first day to emergence with plant height (r=-0.87**) and above-ground biomass (r=-0.84**) was observed, indicating that the chickpea genotypes which emerged sooner produced greater plant size. Seed size and density were found to have no relationship with plant size. Kabuli types on average germinated faster and produced larger plants as opposed to the Desi types under the limited soil moisture content. Susceptibility of the genotypes to limited soil moisture condition was shown through relatively longer delays in time to emergence (lower germination rate) and reduction in seedling parameters as compared to the resistant genotypes. Final average above-ground biomass (plant size) and plant height under the limited soil moisture content, as opposed to adequate moisture level (F. C. 25% vs. 100%), were reduced 79–85% in Kabuli and 77–79% in Desi types, respectively. [10]

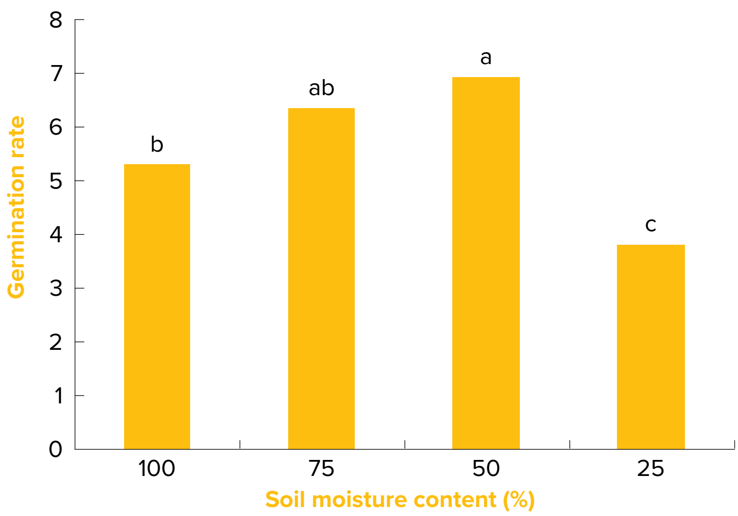

Figure 3: *Effect of soil moisture content (% field capacity basis) on germination rate (averaged over replications and genotypes).* [11]

10 Majnoun Hosseini, N., Siddique, K. H. M., Palta, J. A., & Berger, J. (2009). Effect of soil moisture content on seedling emergence and early growth of some chickpea (*Cicer arietinum* L.) genotypes. Journal of Agricultural Science and Technology, 11, 401-411.

11 Majnoun Hosseini, N., Siddique, K. H. M., Palta, J. A., & Berger, J. (2009). Effect of soil moisture content on seedling emergence and early growth of some chickpea (*Cicer arietinum* L.) genotypes. Journal of Agricultural Science and Technology, 11, 401-411.

Table 1: *Mean soil moisture effects on chickpea genotypes characteristics (averaged over genotypes and replications).* [12]

Soil moisture (%)	Emer-gence (%)	Time to emer-gence	Plant height (cm)	Branch no. per plant	Leaf area cm^2	Above-ground biomass (g $plant^{-1}$)	Specific leaf areas (cm^2 g^{-1})
100	78.4	3.8	20.2	4.5	102.6	0.98	178
75	86.4	6.2	19	4.4	81.8	0.83	164.8
50	83.7	7.9	14.4	3.5	41.6	0.54	131.3
25	56.5	13.9	3.7	1	2.5	0.21	37.3
l.s.d. (P = 0.05)	10.8	1.06	2.3	0.4	23.3	0.2	11.4

For more information on the effects of drought stress, see Section 14: Environmental issues.

4.2 Effect of temperature, photoperiod, climate effects on plant growth and physiology

During their growth, crop plants are usually exposed to different environmental stresses which limit their growth and productivity.

Figure 4 shows crop biomass is driven by:

- the capacity of roots to capture water and nutrients, chiefly nitrogen and phosphorus (black arrow in Figure 4);
- the capacity of canopies to capture radiation and carbon dioxide used in photosynthesis (green arrow in Figure 4);
- the efficiency of the crop to transform resources (water, nutrients, radiation, carbon dioxide) into dry matter (red arrow in Figure 4).

Crop growth and yield depends on the ability of crops to capture above ground and soil resources, and on the capacity of crops to transform these resources into biomass. Environmental factors, such as ambient temperature or soil salinity, modulate the rate of capture of resources and the efficiency in the transformation of resources into plant biomass and these are illustrated by the dashed lines in Figure 4. [13]

12 Majnoun Hosseini, N., Siddique, K. H. M., Palta, J. A., & Berger, J. (2009). Effect of soil moisture content on seedling emergence and early growth of some chickpea (*Cicer arietinum* L.) genotypes. Journal of Agricultural Science and Technology, 11, 401-411.

13 V Sandras, G McDonald. GRDC. (2012). Water Use Efficiency of grain crops in Australia: principles, benchmarks and management.

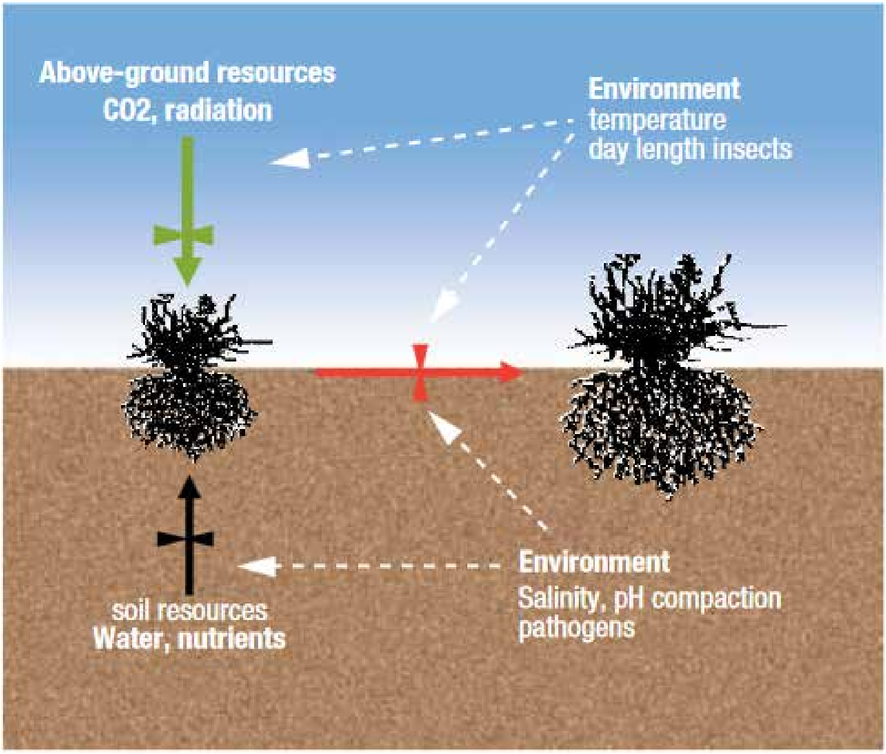

Figure 4: *Factors that drive crop biomass.*

Source: GRDC.

Temperature, day length, and drought are the three major factors affecting flowering in chickpeas. Temperature is generally more important than day length.

4.2.1 Temperature

The timing of flowering is an important trait affecting the adaptation of crops to low-rainfall, Mediterranean-type environments (such as southern Australia), and seed yields of many crops in these areas have been increased by early sowing and the development of early-flowering varieties.

Cold temperatures

Air temperature and photoperiod have a major influence on the timing of reproductive events in chickpeas, with the rate of progress to flowering being a linear function of mean temperature. Flowering is invariably delayed under low temperatures but more branching occurs. [14]

Crop duration is highly correlated with temperature, such that crops will take different times from sowing to maturity under different temperature regimes. Chickpeas, unlike other cool season legumes, are very susceptible to cold conditions, especially at flowering, and any advantage derived from early flowering is often negated by increased flower and pod abortion.

Experiments have shown that the average day/night temperature is critical for flowering and pod set, rather than any specific effects of maximum or minimum temperatures. Pods at a later stage of development are generally more resistant to frost than flowers and small pods, but may suffer some mottled darkening of the seed coat.

The critical mean or average daily temperature for abortion of flowers in most current varieties is <15°C. Abortion occurs below this temperature because the pollen

14 Pulse Australia. Chickpea Production: Southern and Western Region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

becomes sterile and reproductive structures do not develop. Flowers may develop below this temperature but they contain infertile pollen. [15]

In many chickpea crops, it is not until temperatures rise in late August and September that pod set and seed-filling commence. When temperatures rise, true flowers develop within 3–4 days. Even after the production of true flowers, periods of low temperature may result in further flower and pod abortion at intermittent nodes on the stems.

Pollen germination and vigour is also affected by chilling range temperatures. [16]

Sub-zero temperatures in winter and spring can damage leaves and stems of the plant. Frosts can cause bleaching of leaves, especially on the margins, and a characteristic 'hockey-stick' bend in the stem (Photo 4). However, chickpeas have an excellent ability to recover from this superficial damage and is able to regenerate new branches in severe cases.

Photo 4: *Frost can cause bends like a hockey stick in chickpea stems.*

Photo S. Loss, DAFWA.

Late frosts also cause flower, pod, and seed abortion (Photo 5). Pods at a later stage of development are generally more resistant to frost than flowers and small pods, but may suffer some mottled darkening of the seed coat. Empty pods appear bleached and are known as "ballons". They pop when squeezed.

15 Pulse Australia. Chickpea Production: Southern and Western Region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

16 Croser, J. S., Clarke, H. J., Siddique, K. H. M., & Khan, T. N. (2003). Low-temperature stress: implications for chickpea (*5.6* L.) improvement. Critical Reviews in Plant Sciences, 22(2), 185-219.

Photo 5: *Frost can cause pod abortion (usually low on the stem) but the plant may set many pods late in the season if conditions are favourable.*

Photo: T. Knights, NSW DPI.

NOTE: The impacts of low air temperatures will be moderated by topography and altitude, i.e. there will be warmer and cooler areas in undulating country.

For more information, see Section 14: Environmental issues.

Heat stress

Chickpea in its reproductive stage is sensitive to heat stress (32/20°C or higher as day/night temperatures) with consequent substantial loss of potential yields at high temperatures. Temperatures >35°C in spring may also reduce yield in chickpea, causing flower abortion and a reduction in the time available for seed filling. Chickpea, however, is considered more heat-tolerant than many other cool-season grain legumes. The anthers of heat sensitive genotypes have been found to have reduced synthesis of sugars due to inhibition of the appropriate enzymes. Consequently, affected plant pollen can have considerably lower sucrose levels resulting in reduced pollen function, impaired fertilisation, and poor pod set in the heat sensitive genotypes. [17]

In Australia, drought stress often accompanies high temperatures in spring, causing the abortion of flowers, immature pods, and developing seeds. On the other hand, high levels of humidity and low light also prevent pod set.

Chickpea pollen grains are more sensitive to heat stress than the stigma. High temperatures have been found to reduce pollen production per flower, amount of pollen germination, pod set, and seed number. [18]

High air temperatures during the period from flowering to maturity have also been found to reduce the time to maturity of late-sown chickpea and lead to reduced seed size and lower yields. [19]

Chickpea can tolerate high temperature if there is adequate soil moisture, and it is usually one of the last grain legume crops to mature in Mediterranean-type environments.

For more information, see Section 14: Environmental issues.

17 Kaushal, N., Awasthi, R., Gupta, K., Gaur, P., Siddique, K. H., & Nayyar, H. (2013). Heat-stress-induced reproductive failures in chickpea (*Cicer arietinum*) are associated with impaired sucrose metabolism in leaves and anthers. Functional Plant Biology, 40(12), 1334-1349.

18 Devasirvatham, V., Gaur, P. M., Mallikarjuna, N., Tokachichu, R. N., Trethowan, R. M., & Tan, D. K. (2012). Effect of high temperature on the reproductive development of chickpea genotypes under controlled environments. Functional Plant Biology, 39(12), 1009-1018.

19 Sivakumar, M. V. K., & Singh, P. (1987). Response of chickpea cultivars to water stress in a semi-arid environment. Experimental agriculture, 23(01), 53-61.

4.2.2 Photoperiod

Photoperiod is one of the major environmental factors determining time to flower initiation and first flower appearance in plants. In chickpea, photoperiod sensitivity (expressed as delayed to flower under short days (SD) as compared to long days (LD)) may change with the growth stage of the crop. Chickpea is a photoperiod sensitive, long-day plant, where flowering is delayed as day length becomes shorter than a base photoperiod (17 h). Progress towards flowering is rapid during long days (17+ h) and flowering is delayed but never prevented under short day (<17 h) conditions. [20]

IN FOCUS

Determination of Photoperiod-Sensitive Phase in Chickpea (*Cicer arietinum* L.).

For one day-neutral cultivar, there was no significant difference in the number of days to flowering of the plants grown under SD and LD as well as subsequent transfers. In photoperiod-sensitive cultivars, three different phenological phases were identified: a photoperiod-insensitive pre-inductive phase, a photoperiod-sensitive inductive phase, and a photoperiod-insensitive post-inductive phase.

The photoperiod-sensitive phase extends after flower initiation to full flower development. Results from this research will help to develop cultivars with shorter pre-inductive photoperiod-insensitive and photoperiod-sensitive phases to fit to regions with short growing seasons. [21]

4.2.3 Water and moisture

About 90% of chickpeas in the world are grown under rainfed conditions where drought is one the major constraints, limiting its production. Drought affects various morphological and physiological processes, resulting in reduced growth, development and economic yield of crop. Water stress has prominent effect on leaf number, total leaf area, and secondary branches, causing invariable reduction under rainfed conditions. Several studies have shown that optimum yield can be obtained by irrigation at branching, flowering, and pod formation stages.

The reactions of plants to water stress vary depending upon intensity and duration of stress, as well as plant species and its stage of growth. Stress during vegetative phase reduce grain yield through reducing plant size, restricting leaf area, dry matter accumulation and limiting number of pods. However, water deficits at the flowering and the post flowering stages have been found to have greater adverse impact than at the vegetative stage. [22]

Starting soil water can have a strong influence on the yield expectation of chickpea as well as the riskiness of production.

Yields are best in areas with reliable winter rainfall for crop growth and mild spring conditions during seed filling. Chickpea is well suited to well-drained, non-acidic soils with medium to heavy clay texture. [23]

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20 Pulse Australia. Chickpea Production: Southern and Western Region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

21 Daba, K., Warkentin, T. D., Bueckert, R., Todd, C. D., & Tar'an, B. (2016). Determination of Photoperiod-Sensitive Phase in Chickpea (*Cicer arietinum* L.). Frontiers in plant science, 7.

22 Randhawa, N., Kaur, J., Singh, S., & Singh, I. (2014). Growth and yield in chickpea (*Cicer arietinum* L.) genotypes in response to water stress. African Journal of Agricultural Research, 9(11), 982-992.

23 Pulse Australia. Chickpea Production: Southern and Western Region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

Soon after the development of pods and seed-filling, senescence of subtending leaves begins. If there is plenty of soil moisture and maximum temperatures are favourable for chickpea growth, flowering and podding will continue on the upper nodes. However, as soil moisture is depleted, flowering ceases and eventually the whole plant matures. This is typical of grain legumes and annual plants in general.

Research has indicated that unlike other winter pulses under mild moisture stress, chickpeas are capable of accumulating solutes (sugar, proteins, and other compounds) in their cells, thereby maintaining stomatal conductance and low levels of photosynthesis. This process is known as osmoregulation.

Chickpeas can access moisture to 90 cm depth provided there is no compaction or saline/sodic layers or boron in the soil profile.

IN FOCUS

Growth and yield in chickpea (*Cicer arietinum* L.) genotypes in response to water stress.

Twenty chickpea genotypes were grown under rainout shelter to investigate the influence of water stress treatments imposed at varied growth stages.

The maximum reduction in height and branches was observed when irrigation was restricted at T2 stage. Restricted irrigation decreased the biomass of stem, leaves and roots leading to reduced leaf area and leaf area index as well. The yield traits viz. 100 seed weight, total number of pods, percentage filled pods were reduced significantly under stress. The grain yield under restricted conditions was reduced by 40.50 to 55.91% over irrigated control in T4 to T2, respectively. [24]

4.2.4 Drought stress

Chickpea has been shown to be one of a number of pulses that is suited to the fine textured, neutral-to-alkaline soils of the eastern cropping zone of southern Australia. Pulses are subjected to terminal drought in this environment. Studies have shown that pollination and pod development are inhibited by low temperatures in chickpea, thus under the winter conditions present in southern Australia, pod set and seed filling are delayed until spring when leaf photosynthetic rates are low as a consequence of soil water depletion.

One of the major consequences of this is that terminal drought reduces the size of the seed, particularly in late-formed seeds. As seed size and uniformity are important in determining the market price, particularly in Kabuli chickpeas, any variation among genotypes in maintaining seed size under conditions of terminal drought will be important in breeding for improved yield and quality in chickpeas for drought-prone environments.

24 Randhawa, N., Kaur, J., Singh, S., & Singh, I. (2014). Growth and yield in chickpea (*Cicer arietinum* L.) genotypes in response to water stress. African Journal of Agricultural Research, 9(11), 982-992.

IN FOCUS

Seed growth of Desi and Kabuli chickpea in a short-season Mediterranean-type environment

The influence of terminal drought on the seed growth of three chickpea genotypes was examined in a field experiment in south-western Australia. Tyson, a small-seeded Desi cultivar, ICCV88201, a Desi breeding line (sister line to the recently released Sona cultivar) with medium-sized seed, and Kaniva, a Kabuli cultivar with large seed, were grown under rainfed and irrigated conditions.

Genotypic differences in the maximum rate of seed fill were found to exist in chickpea. Both the rate and duration of seed growth were reduced in the rainfed plants, regardless of genotype. Reductions in the dry weight of the pod shell suggest that the remobilisation of dry matter from the pod may contribute 9–15% of the seed weight in rainfed chickpea. [25]

For more information about factors affecting chickpea growth, see Section 14: Environmental Issues.

4.3 Plant growth stages

Photo 6: *Newly established chickpea plant.*

ABC Rural, Jodie Gunders.

The chickpea crop germinates, matures, senesces, and dies within 100–225 days from sowing, depending on environmental conditions before and after flowering, the magnitude of seed yield, and the rate and synchrony of seed filling (Photo 6). [26]

25 Davies, S. L., Turner, N. C., Siddique, K. H. M., Leport, L., & Plummer, J. A. (1999). Seed growth of desi and kabuli chickpea (*Cicer arietinum* L.) in a short-season Mediterranean-type environment. Animal Production Science, 39(2), 181-188.

26 Croser, J. S., Clarke, H. J., Siddique, K. H. M., & Khan, T. N. (2003). Low-temperature stress: implications for chickpea (*Cicer arietinum* L.) improvement. Critical Reviews in Plant Sciences, 22(2), 185-219.

Photo 7: *Chickpea growth and development from germination to two months. Plants may vary according to variety and environment.*

Photo: H. Clarke, UWA.

The chickpea plant is erect and freestanding, usually 15–60 cm in height, although well-grown plants may grow to 80 cm. The plants have a fibrous taproot system, a number of woody stems forming from the base, upper secondary branches and fine, frond-like leaves. Chickpeas are considered very indeterminate in their growth habit; i.e. their terminal bud is always vegetative and keeps growing, even after the plant switches to reproductive mode and flowering begins. [27]

Early research indicates that chickpea seed yield can be reduced by 81% and straw yields by 63% when fields remained weed infested until harvest, compared with weed-free conditions throughout the growing season. The critical period of weed interference has been suggested to be between 35 and 49 days after emergence in chickpea. [28]

Another study expanded on this research and found that chickpea must be kept weed-free between the five-leaf and full flowering stages (24–48 DAE) and from the four-leaf to beginning of flowering stages (17–49 DAE) in order to prevent >10% seed yield loss. [29]

The chickpea growth stages key is based on counting the number of nodes on the main stem (Table 2). Uniform growth stage descriptions were developed for the chickpea plant based on visually observable vegetative (V) and reproductive (R) events. The V stage was determined by counting the number of developed nodes on the main stem, above ground level. The last (uppermost) node counted must have its leaves unfolded. The R stages proposed begin when the plant begins to flower at any node.

27 Pulse Australia. Chickpea Production: Southern and Western Region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

28 Al-Thahabi, S. A., Yasin, J. Z., Abu-Irmaileh, B. E., Haddad, N. I., & Saxena, M. C. (1994). Effect of weed removal on productivity of chickpea (*Cicer arietinum* L.) and lentil (Lens culinaris Med.) in a Mediterranean environment. Journal of Agronomy and Crop Science, 172(5), 333-341.

29 Mohammadi, G., Javanshir, A., Khooie, F. R., Mohammadi, S. A., & Zehtab Salmasi, S. (2005). Critical period of weed interference in chickpea. Weed research, 45(1), 57-63.

Table 2: *Growth stages of a chickpea plant (Nolan 2001).*

Designation	Growth stage	Description
Vegetative growth stage (V-stage) in chickpeas		
VG	Germination	Cotyledons remain underground inside the seed coat and provide energy for rapidly growing primary roots (radicle) and shoots
VE	Emergence	The plumule emerges and the first two leaves are scales. The first true leaf has two or three pairs of leaflets plus a terminal leaflet
V1	First node	Imparipinnate (terminal unpaired) leaves attached to the first node are fully expanded and flat while the 1st imparipinnate leaf attached to the upper node starts to unroll
V2	Second node	1st imparipinnate leaf attached to the second node is fully expanded and flat while the 2nd imparipinnate leaf on the upper node starts to unroll
V3	Third node	2nd imparipinnate leaf attached to the third node is fully expanded and flat while the 3rd imparipinnate leaf on the upper node starts to unroll. The bulk of the yield is found on the branches stemming from the first three nodes
V(n)	N-node	A node is counted when its imparipinnate leaf is unfolded and its leaflets are flat
Reproductive growth stage (R-stage) in chickpea		
R0	False flowering	In the transition from vegetative to include reproductive growth, a number of false flowers (called pseudo flowers) may develop from the axillary buds. These flower buds lack fully developed petals and typically appear if flowering is triggered before mean temperatures are high enough for true flowers to develop, especially if soil has high moisture content coinciding with flowering, which enables it develop a bigger canopy
R1	Start flowering	One flower bud at any node on the main stem (see p. 5 in 'The chickpea book', Loss *et al.* 1988)
R2	Calyx opening	Bud grows but is still sterile, sepals begin to form
R3	Anthesis	Pollination occurs before the bud opens
R4	Wings extend	Flower petals extend to form a flower
R5	Corolla collapses	Flower collapses and petals senesce and peduncle reflexes so that the developing pod usually hangs below its subtending leaf
R6	Pod initiation	One pod is found on any node on the main stem
R7	Full pod	One fully expanded pod is present that satisfies the dimensions characteristic of the cultivar
R8	Beginning seed	One fully expanded pod is present in which seed cotyledon growth is visible when the fruit is cut in cross-section with a razor blade. (Following the liquid endosperm stage)
R9	Full seed	One pod with cavity apparently filled by the seeds when fresh
R10	Beginning maturity	One pod on the main stem turns to a light golden-yellow in colour

Designation	Growth stage	Description
R11	50% golden pod	50% of pods on the plant mature
R12	90% golden pod	90% of pods physiologically mature (golden yellow), usually about 140–200 days after planting depending on season and cultivar

For populations, vegetative stages can be averaged if desired. Reproductive stages should not be averaged.

A reproductive stage should remain unchanged until the date when 50% of the plants in the sample demonstrate the desired trait of the next reproductive (R) stage. The timing of a reproductive stage for a given plant is set by the first occurrence of the specific trait on the plant, without regard to position on the plant (Photo 8). [30]

Photo 8: *Growth habit of a chickpea plant.*

4.3.1 Leaves

Leaves in chickpeas are alternate along the branch (Photo 9). The first true leaf has two or three pairs of leaflets plus a terminal one. Fully formed leaves, with 5–8 pairs of serrated leaflets (10–16 leaflets), usually develop after the sixth branch (node) stage. Leaflets can fold slightly in dry conditions to minimise transpiration. Despite having more leaves and branches than other legume crops such as faba beans, canopy development in chickpeas is slow, especially during the cool winter months.

30 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited

Photo 9: *Alternate leaves along the branch, with multiple leaflets on each leaf.*

Photo: G. Cumming, Pulse Australia

The entire surface of the plant shoot, except the flower, has a thick covering of glandular hairs (trichomes) that secrete a strong acid (mostly malic acid), particularly during pod-set (Photo 10). The malic secretions from all vegetative surfaces of the plant seem to play a role in protecting the plant against pests such as red-legged earth mite, lucerne flea, aphids, and pod borers. Similar substances are also secreted from the root system and can solubilise soil-bound phosphate and other nutrients. The acid also corrodes leather boots.

Photo 10: *Green pods covered in glandular hairs excreting acid.*

Source: LloydslistAus.

4.3.2 Roots

Chickpea root systems are usually deep and strong, and contribute to the ability to withstand dry conditions. The plant has a taproot with few lateral roots. Root growth is most rapid before flowering but will continue until maturity under favourable conditions. Although rare, in deep well-structured soils, roots can penetrate more than 1 m deep (Photo 11); however, subsoil constraints such as soil chloride >800 mg/kg soil in the top 60 cm will restrict root growth and water availability.

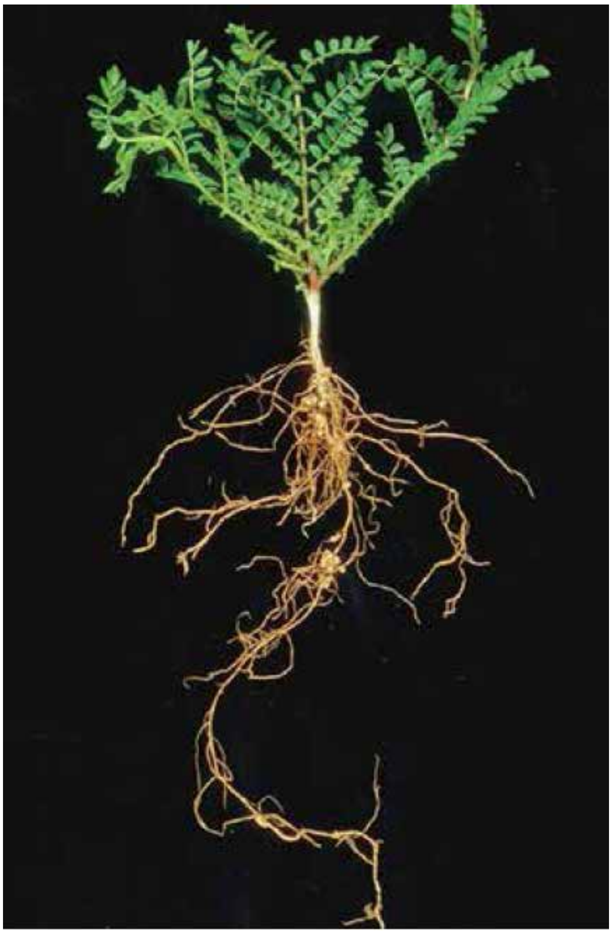

Photo 11: *Chickpea usually has a deep tap root system.*

Photo: P. Maloney, DAFWA.

As well as their role in water and nutrient uptake, chickpea roots develop symbiotic nodules with the Rhizobium bacteria, capable of fixing atmospheric nitrogen. The plant provides carbohydrates for the bacteria in return for nitrogen fixed inside the nodules. Chickpea plants can derive more than 70% of their nitrogen requirement from symbiotic nitrogen fixation.

These nodules are visible within about a month of plant emergence, and eventually form slightly flattened, fan-like lobes (Photo 12). Practically all nodules are confined to the top 30 cm of soil and 90% are within the top 15 cm of the profile. When cut open, nodules actively fixing nitrogen have a pink centre. Nitrogen fixation is highly sensitive to waterlogging so it is essential that chickpea crops are grown on well-aerated and drained soils. [31]

31 Pulse Australia. Chickpea Production: Southern and Western Region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

Photo 12: *Well-nodulated chickpea plants.*

Photo: G. Cumming, Pulse Australia

4.3.3 Branches

Primary branches, starting from ground level, grow from buds at the lowest nodes of the plumular shoot as well as the lateral branches of the seedling. These branches are thick, strong, and woody, and they determine the general appearance of the plant (Figure 5). The main stem and branches can attain a height of about 40–100 cm. Kabuli varieties are generally taller than Desi varieties.

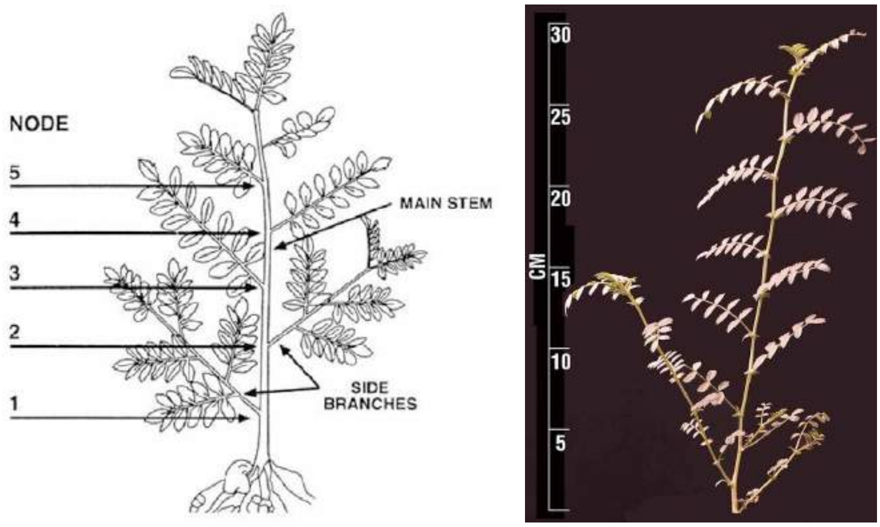

Figure 5: *Chickpea at the 5–7-node stage of development, prior to flowering.*

Secondary branches are produced by buds on the primary branches. They are less vigorous but contribute to a major proportion of the plant yield. Tertiary branches growing from buds on secondary branches are more leafy and carry fewer pods. The number of primary branches can vary from one to eight depending upon the variety and growing conditions. In chickpeas, five branching habits based on angle of branches from the vertical are classified: erect, semi-erect, semi-spreading, spreading, and prostate. Most modern varieties are erect or semi-erect, to enable mechanical harvesting. The final height of the plant is highly dependent on environmental conditions and the variety being grown, but in general, can range from 50 to 100 cm. [32]

4.3.4 Flowering

Growth in chickpeas is often described as 'indeterminate'. This means that branch and leaf (or vegetative) growth continues as the plant switches to a reproductive mode and initiates flowering. Hence, there is often a sequence of leaf, flower bud, flower and pod development along each branch (Photo 13).

The onset and duration of flowering in chickpea are functions of genotype, photoperiod, and temperature. Flowering is indeterminate and can extend for up to 60 days with leaf initiation and stem elongation continuing into the reproductive period. [33]

Photo 13: *Different stages of flower development on the same chickpea branch.*

Photo K. Siddique, DAFWA.

Chickpea is peculiar among pulses in that a number of pseudo-flowers or false flower buds develop during the changeover from leaf buds to flower buds on the stem. Therefore, there could be a period of ineffective flowering when pod set does not occur.

In warmer tropical and subtropical environments, this period is minimal but in cooler temperate–subtropical environments, it can be as long as 50 days. Flowering commences on the main stem and lower branches and proceeds acropetally at intervals averaging 1.5–2 days between successive nodes along each branch. The bulk of the yield is found on the branches stemming from the first three nodes.

The fruit develops in an inflated pod containing 2–4 ovules, of which one or two usually develop into seeds. At any location, seasonal variations in temperature can bring about a significant shift in flowering times (i.e.±10 days from the figures quoted

32 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

33 Croser, J. S., Clarke, H. J., Siddique, K. H. M., & Khan, T. N. (2003). Low-temperature stress: implications for chickpea (*Cicer arietinum* L.) improvement. Critical Reviews in Plant Sciences, 22(2), 185-219.

below). In general, warmer temperatures hasten development. Chickpeas will tolerate higher temperatures during flowering than peas or lupins. Cool wet conditions at flowering can adversely affect seed set.

Petals are generally purple in the Desi type and white-to-cream in the Kabuli type (Photo 14). Purple-flowered Desi types generally contain high amounts of the red pigment anthocyanin, and their leaves, stems and seed coats are generally dark. By contrast, the white-flowered Kabuli types lack anthocyanin, have light green leaves and stems, and pale seeds. Increased pigmentation is evident following environmental stresses such as low temperature, salinity, waterlogging, drought, and virus infection, especially in Desi types.

Photo 14: *Desi chickpea purple flower (left) and Kabuli chickpea flower (right). Kabuli chickpeas lack anthocyanin, hence their white flowers.*

Photos: G. Cumming, Pulse Australia.

Pollination takes place before the flower bud opens in chickpea, when the pollen and the receptive female organ are still enclosed within a fused petal, called the keel (Table 3 and Photo 15). Natural crosspollination has been reported; however, most studies indicate 100% self-pollination.

Table 3: *Stages of pollen development in chickpea grown at 25/18°C day/night temperatures (12 hour daylength).* [34]

Description of bud	Time before anthesis (days)	Stage of pollen development
Microscopic bud (<0.5 mm)	9	Pre-meiotic microspore mother cells
Very small bud (0.75 mm)	7-8	Early stages of first division meiosis
Small bud (1 mm)	6	Late first division, second division meiosis and early tetrads
Small bud (1.5 mm)	4-5	Late tetrads and microspore release
Sepals much larger than petals	3	Vacuoles appear and mitosis occurs; generative and vegetative nuclei and sperm cells visible
Sepals slightly larger than petals (petal visible)	2	Maturation, starch accumulation
Sepals same length at petals	1	Mature pollen, vacuoles disappear
Hooded flower	Anthesis	Mature dry pollen

34 Clarke, H. J., & Siddique, K. H. M. (2004). Response of chickpea genotypes to low temperature stress during reproductive development. Field Crops Research, 90(2), 323-334.

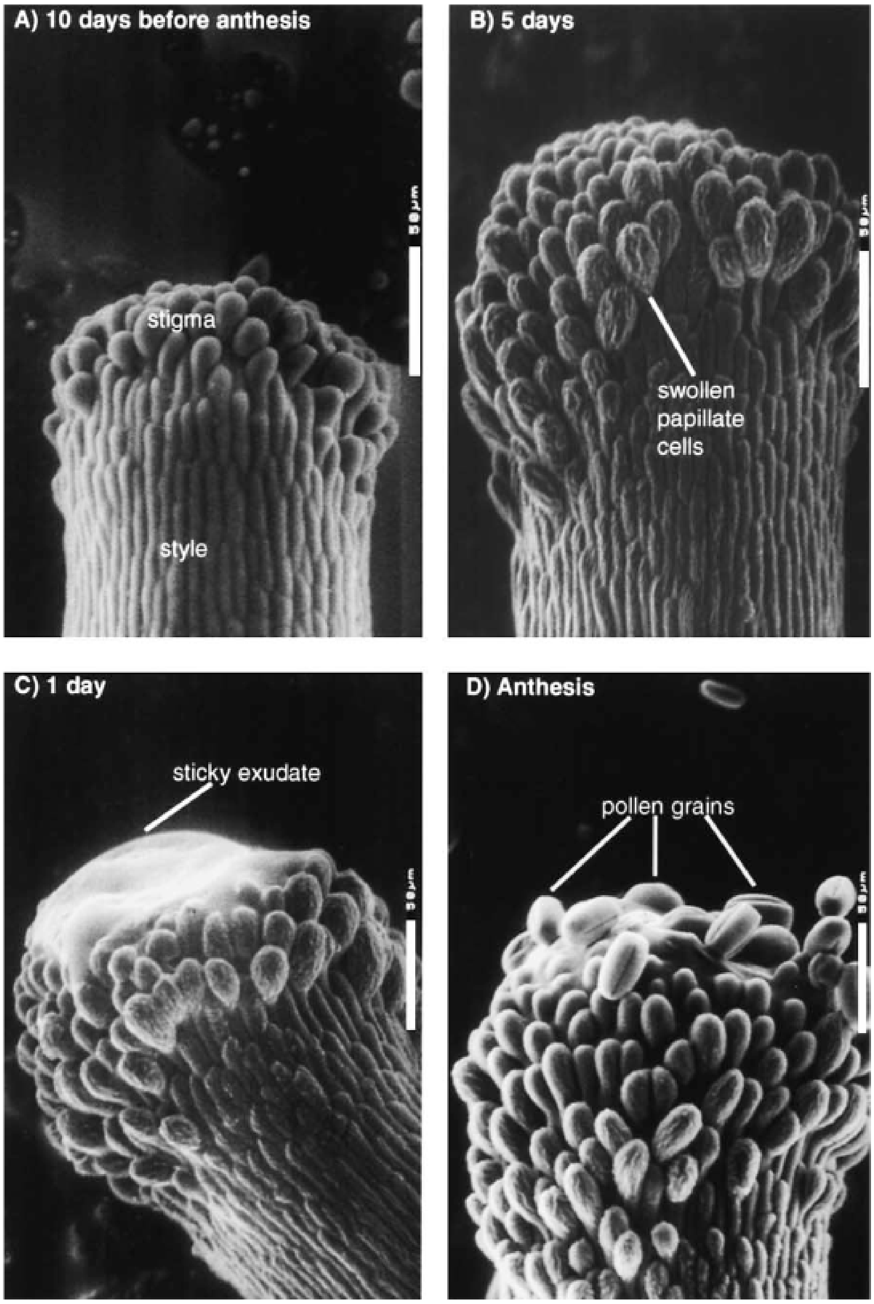

Photo 15: *Development of the style and stigma in chickpea buds from 10 days before anthesis to the fully reflexed flower when plants were grown at 25/18°C, 12 hour daylength. A low temperature regime of 12/7°C increased the time for the same development from 10 to 20 days.* [35]

Flower terminals normally develop from the axillary bud at the base of each node. Flowers are borne on a jointed peduncle that arises from nodes. Flowers are primarily self-pollinated, with most reports measuring 100% self-pollination.

Chickpea plants generally produce many flowers. However, ~30% do not develop into pods, depending upon the variety, sowing date and other environmental conditions.

35 Clarke, H. J., & Siddique, K. H. M. (2004). Response of chickpea genotypes to low temperature stress during reproductive development. Field Crops Research, 90(2), 323-334.

4.3.5 Podding

Initial stages of seed development involve the pod wall expanding rapidly—in chickpea achieving its maximum dry weight while the embryo is very small and is in a phase of cell division. Seed growth is then characterised by a high rate of metabolic activity associated with the rapid, linear accumulation of dry matter, principally as starch and storage protein. At the end of this phase, a period of dehydration and maturation follows, by which time there is little endosperm left, with the embryo filling the seed coat. The maximum potential size of a seed is a function of the rate and duration of embryo growth. Environmental factors such as temperature and water availability affect seed growth rate and final seed size. [36]

Under favourable temperature and soil moisture conditions, the time taken from fertilisation of the ovule (egg) to the first appearance of a pod (pod set) is about six days (Figure 6). The seed then fills over the next 3–4 weeks (Figure 7). Once a pod has set, the jointed peduncle of the senescing petals reflexes, so that the developing pod hangs beneath its subtending leaf. After pod set, the pod wall grows rapidly for the first 10–15 days, and seed growth mainly occurs later.

Figure 6: *Chickpea podding (left) and chickpea plant seven weeks before harvest (right).*

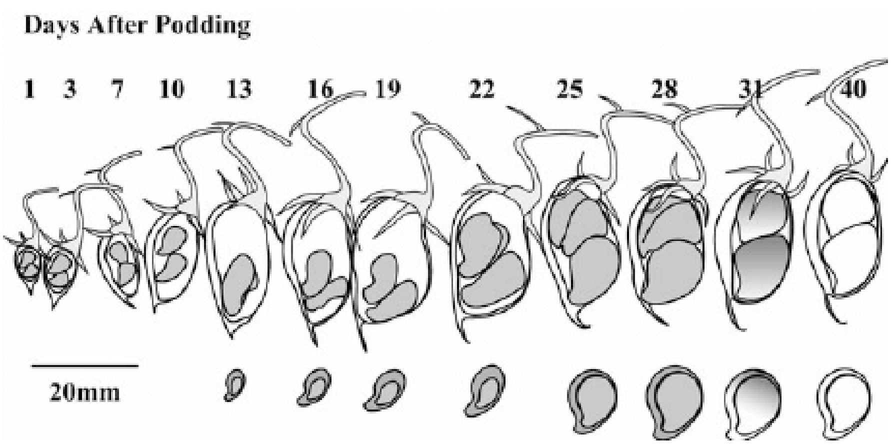

Figure 7: *Seed and pod development in chickpeas, showing the relative sizes of the pod, seed coat, embryo, and the internal pod gas volume.* [37]

36 Davies, S. L., Turner, N. C., Siddique, K. H. M., Leport, L., & Plummer, J. A. (1999). Seed growth of desi and kabuli chickpea (*Cicer arietinum* L.) in a short-season Mediterranean-type environment. Animal Production Science, 39(2), 181-188.

37 Furbank, R. T., White, R., Palta, J. A., & Turner, N. C. (2004). Internal recycling of respiratory CO_2 in pods of chickpea (*Cicer arietinum* L.): the role of pod wall, seed coat, and embryo. Journal of Experimental Botany, 55(403), 1687-1696.

Chickpea pods vary greatly in size between varieties. Pod size is largely unaffected by the environment. By contrast, seed filling and subsequent seed size are highly dependent on variety and weather conditions.

Seeds are characteristically 'beaked', sometimes angular, with a ridged or smooth seed coat. Seed colour varies between varieties from chalky white to burgundy and brown, to black, and is determined by the colour and thickness of the seed coat and the colour of the cotyledons inside. Seeds vary from one to three per pod.

In southern Australia, chickpea crops can reach maturity 140–200 days after sowing, depending on the sowing date, variety, and a range of environmental factors including temperature. Chickpeas become ready to harvest when 90% of the stems and pods lose their green colour and become light golden-yellow. At this point, the seeds are usually hard and rattle when the plant is shaken (Photo 16). [38]

Photo 16: *Physiologically mature grains 'rattle pod'.*

Photo: G. Cumming, Pulse Australia.

38 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

FAQ

Nutrition and fertiliser

Key messages

- Incorrect levels of nutrients (too little, too much, or the wrong proportion) can cause nutritional problems.
- Today, the main method to maintain or restore soil nutrients and increase crop yields is the application of mineral fertilisers.
- A soil or plant tissue test will help to identify what nutrients are limiting yield or quality.
- Become familiar with plant and paddock symptoms of various nutritional deficiencies.
- If chickpea plants have effectively nodulated, they should not normally need N fertiliser.
- Phosphorus is the main nutrient of consideration. Trace elements zinc, manganese and iron are always important to production and quality but remain variable and as such should be evaluated by soil and plant tests, local experience and paddock trials.
- Molybdenum and cobalt are required for effective nodulation and should be applied as needed. Foliar sprays of zinc and manganese may be needed where deficiencies of these micronutrients are a known problem, in particular on high-pH soil types.

Incorrect levels of nutrients (insufficient, excess or disproportionate) can cause nutritional deficiencies or toxicities. If the condition is extreme, plants will show visible symptoms that can sometimes be identified. Visual diagnostic symptoms are readily obtained and they provide an immediate evaluation of nutrient status. Visual symptoms do not develop until a major effect on yield, growth or development has occurred; therefore, damage can be done before there is visual evidence of it.

Healthy plants are more able to ward off disease, pests, and environmental stresses and so achieve higher yield and better grain quality. [1] Ensuring adequate nutrition will assist the chickpea crop to generate dense uniform canopies, which deter aphids. [2]

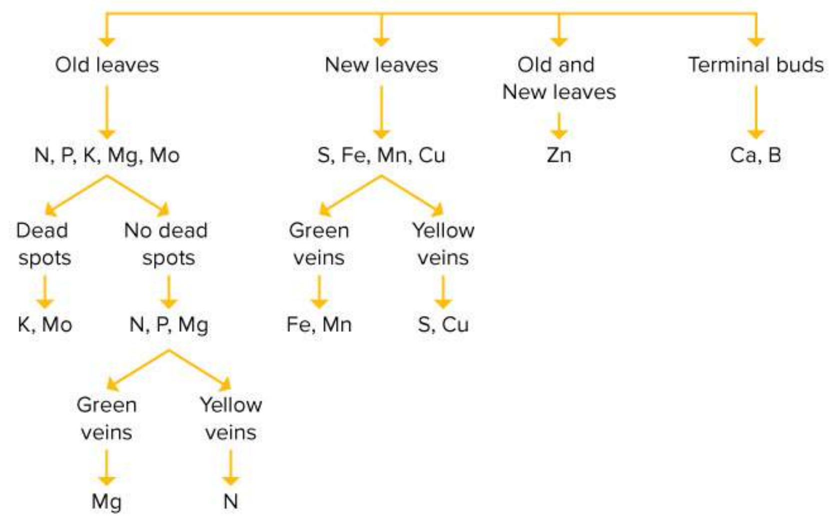

Figure 1: *Flow chart for the identification of deficiency symptoms.*

Source: T Reddy, G Reddi, Kalyani Publishers

1 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

2 A Verrell (2103) Wirus in chickpea in northern NSW 2012. GRDC Update Papers 26 March 2013, http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/02/Virus-in-chickpea-in-northern-NSW-2012

In south-eastern Australia profitable grain production depends on applied fertilisers, particularly nitrogen (N), phosphorus (P) and to a lesser extent, potassium (K), sulfur (S), zinc (Zn), manganese (Mn) and copper (Cu).

The more attention paid to all of the activities that contribute to nutrient management (Figure 2), the better the outcome achieved from soil and plant testing. Testing may not provide a useful contribution if one or more of these steps is not done well.

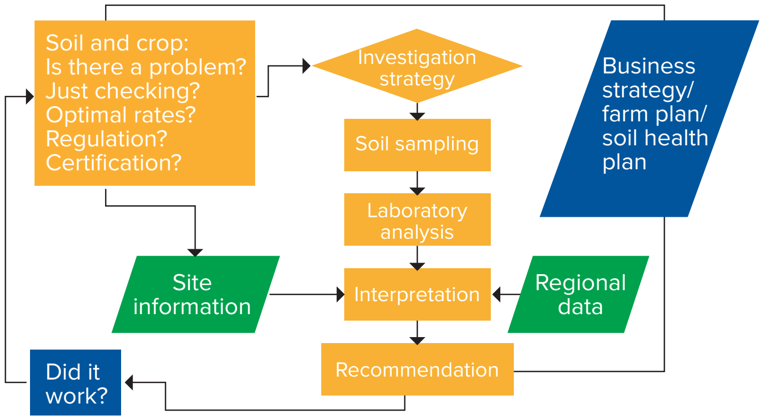

Figure 2: *Nutrient management flow chart.* [3]

5.1 Nutrient types

Plant nutrients are categorised as either macronutrients or micronutrients (also called trace elements). Macronutrients are those elements that are needed in relatively large amounts. They include N, P, and K, which are the primary macronutrients, with calcium (Ca), magnesium (Mg), and sulfur (S) considered as secondary. Higher expected yields of crops for grain or forage will place greater demand on the availability of major nutrients such as P, K, and S. Nitrogen, P, and at times S are the main nutrients commonly lacking in Australian soils. Others can be lacking under certain conditions. Keep in mind that each pulse type is different, with different requirements for nutrients and may display different symptoms of deficiency. A balance sheet approach to fertiliser inputs is often a good starting point when determining the amount and type (analysis) of fertiliser to apply. Other factors such as a soil test, paddock history, soil type, and personal experience are important inputs to the decision process. Tissue analysis can be helpful in identifying deficiencies once the crop is growing, and can assist in fine-tuning nutrient requirement even when deficiency symptoms are not visible. Micronutrients are those elements that plants need in small amounts, for example iron (Fe), boron (B), manganese (Mn), zinc (Zn), copper (Cu), chlorine (Cl), and Mo.

Both macro- and micronutrients are taken up by roots and certain soil conditions are required for that to occur. Soil must be sufficiently moist to allow roots to take up and transport the nutrients. Plants that are moisture-stressed from either too little or too much moisture (saturation) can often exhibit deficiencies even though a soil test may show these nutrients to be adequate. Soil pH has an effect on the availability of most nutrients and must be within a particular range for nutrients to be released from soil particles. On acid soils, Aluminium (Al) and Mn levels can become elevated and toxic to plants. Aluminium toxicity disrupts the structure and funtion of plant roots, presenting as stunted roots, limiting nutrient and moisture uptake. Aluminium will form a bond with available soil P making it unavailable to plants in the short to medium

3 GRDC (2013) Better fertiliser decisions for crop nutrition. GRDC Crop Nutrition Fact Sheet November 2013, http://grdc.com.au/Resources/Factsheets/2013/11/Better-fertiliser-decisions-for-crop-nutrition

term. Mn toxicity affects plant development with a range of symptoms including leaf yellowing and tissue death, mainly on older leaves. If Al and Mn levels increase, plant growth can be restricted, usually by limiting rhizobia and therefore the plant's ability to nodulate. Soil temperature must lie within a certain range for nutrient uptake to occur. Cold conditions can induce deficiencies of nutrients such as Zn or P. The optimum range of temperature, pH, and moisture can vary for different pulse species. Thus, nutrients may be physically present in the soil, but not available to those particular plants. Knowledge of a soil's nutrient status (soil test) pH, texture, history, and moisture status can be very useful for predicting which nutrients may become deficient. Tissue tests can help to confirm the plant nutrient status. [4]

5.2 Crop removal rates

If the nutrients (P, N, Zn, etc.) removed as grain from the paddock are not replaced, then soil fertility and crop yields will fall. This means that fertiliser inputs must be matched to expected yields and soil type. The higher the expected yield, the higher the fertiliser input, particularly for the major nutrients P, K, and S. The nutrient removal per tonne (t) of grain of the various pulses is shown in Table 1. Actual values may vary by 30%, or sometimes more, because of differences in soil fertility, varieties, and seasons. For example, P removed by 1 t of faba bean grain can vary from a low 2.8 kg on low-fertility soils to 5.4 kg on high-fertility soils. From the table, a 2 t/ha crop of chickpeas will on average remove ~6.5 kg/ha of P. This then is the minimum amount of P that needs to be replaced. Higher quantities may be needed to build up soil fertility or overcome soil fixation of P.

Table 1: *Nutrients removed by one tonne of chickpea grain.*

Chickpea	Kilograms (kg)					Grams (g)			
	N	P	K	S	Ca	Mg	Cu	Zn	Mn
Desi	33	3.2	9	2.0	1.6	1.4	7	34	34
Kabuli	36	3.4	9	2.0	1.0	1.2	8	33	22

Source: Pulse Australia.

Soil types do vary in their nutrient reserves. For example, most black and red soils have sufficient reserves of K to grow many crops. However, the light, white sandy soils, which, on soil test, have <50 µg/g (ppm) (bicarbonate test) of K, will respond to applications of K fertiliser. Other soils may have substantial nutrient reserves that vary in availability during the growing season or are unavailable due to the soil pH. This can often be the case with micronutrients. Foliar sprays can be used in these cases to correct any micronutrient deficiencies. [5]

5.2.1 Nutrient budgeting

When grain is harvested from the paddock, nutrients are removed in the grain. If, over time, more nutrients are removed than are replaced (via fertiliser), then the fertility of the paddock will fall. Nutrient budgeting is a simple way to calculate the balance between nutrient removal (via grain) and nutrient input (via fertiliser).

Table 2 uses standard grain nutrient analyses from Table 1. For a more accurate guide to nutrient removal, use analysis of grain grown on your farm. A more complete picture emerges when several years of a rotation are budgeted.

4 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

5 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

Table 2: *An example of nutrient budgeting.* [6]

Year	Crop	Yield (t/ha)	Nutrients removed (kg/ha)			
			N	P	K	S
2006	Faba bean	2.2	90	8.8	22	3.3
2007	Wheat	3.8	87	11.4	15	5.7
2008	Barley	4.2	84	11.3	21	6.3
2009	Chickpea	1.8	59	5.8	16	3.6
		Total	320	37.3	74	18.9
Year	**Fertiliser**	**Rate (t/ha)**	**Nutrients applied (kg/ha)**			
			N	P	K	S
2006	0 : 20 :0 (NPK)	50	0	10	0	1
2007	18 : 20 :0 (NPK)	70	12.6	14	0	1
2008	18 : 20 :0 (NPK)	70	12.6	14	0	1
	Urea	60	27.6	0	0	0
2009	0 : 16 :0 :20 (NPK)	80	0	12.8	0	16
		Total	52.8	50.8	0	19
Balance			-267.2	+13.5	-74	0

As can be seen from Table 2, a simple nutrient budget, some interpretation of a nutrient budget is needed:

- Nitrogen: The deficit of 267 kg needs to be countered by any N fixation that occurred. This may have been 50 kg/ha per legume crop. It still shows that the N status of the soil is falling and that it should be increased by using more N in the cereal phase. Estimating N fixation is not easy. One rule to use is 20 kg of N is fixed per tonne of plant dry matter at flowering.
- Phosphorus: The credit of 13 kg will be used by the soil in building P levels, hence increasing soil fertility. No account was made for soil fixation of P.
- Potassium: Some Australian cropping soils (usually white sandy soils) are showing responses to K, and applications should be considered at least to replace the K used by the crop.
- Sulfur: Crop removal of S may exceed inputs.

Other nutrients such as Zn and Cu can also be included in a nutrient-balancing exercise. This is a useful tool for assessing the nutrient balance of a cropping rotation; however, it needs to be considered in conjunction with other nutrient-management tools such as soil and tissue testing, soil type, soil fixation, and potential yields. Because P is the basis of soil fertility and, hence, crop yields, all fertiliser programs are built on the amount of P needed. Table 3 shows the required P rates and the rates of various fertilisers needed to achieve this. Many fertilisers are available to use on pulses; for the best advice, check with your local fertiliser reseller or agronomist.

6 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

Table 3: *Fertiliser application rate ready-reckoner (all rates are kg/ha) for some of the fertilisers used on pulses.*

P	Single 8.6% P		Superphosphate Gold Phos 10 18% P		Triple 20% P		6:16:0:10 Legume Special			10:22:0 MAP		18:20:0 DAP		0:15:0:7 Grain Legume Super	
	Fert.	S	Fert.	S	Fert.	S	Fert.	N	S	Fert.	N	Fert.	N	Fert.	S
10	116	13	50	5	45	0.7	62	4	6	46	5	50	9	69	5
12	140	15	67	7	60	0.9	75	4	8	55	6	60	11	83	6
14	163	18	78	8	70	1.1	87	5	9	64	6	70	13	97	7
16	186	20	89	9	80	1.2	99	6	10	73	7	80	14	110	8
18	209	23	100	10	90	1.4	112	6	11	82	8	90	16	124	9
20	223	25	111	11	100	1.5	124	7	12	91	9	100	18	138	10
22	256	28	122	12	110	1.7	137	8	14	100	10	110	20	152	11
24	279	31	133	13	120	1.8	149	8	15	110	11	120	22	166	12

There is a trend to using 'starter' fertilisers such as mono- and di-ammonium phosphate (MAP and DAP) on pulses. Some growers are concerned that using N on their pulse crop will affect nodulation. This is not the case with the low rates of N supplied by MAP or DAP. A benefit of using the starter N is that early plant vigour is often enhanced, and on low fertility soils, yield increases have been gained. [7]

5.3 Identifying nutrient deficiencies

Many nutrient deficiencies may look similar. To identify deficiencies:

- Know what a healthy plant looks like in order to recognise symptoms of distress.
- Determine what the affected areas of the crop look like. For example, are they discoloured (yellow, red, brown), dead (necrotic), wilted or stunted?
- Identify the pattern of symptoms in the field (patches, scattered plants, crop perimeters).
- Assess affected areas in relation to soil type (pH, colour, texture) or elevation.
- Look at individual plants for more detailed symptoms such as stunting, wilting and where the symptoms are appearing (whole plant, new leaves, old leaves, edge of leaf, veins etc.).

If more than one problem is present, typical visual symptoms may not occur. For example, water stress, disease or insect damage can mask a nutrient deficiency. If two nutrients are simultaneously deficient, symptoms may differ from the deficiency symptoms of the individual nutrients. Micronutrients are often used by plants to process other nutrients or work together with other nutrients, so a deficiency of one may look like deficiency of another. For instance, molybdenum (Mo) is required by pulses to complete the process of nitrogen (N) fixation and the symptoms present as nitrogen deficiency. [8]

See sections below for specific symptoms of each nutrient deficiency.

5.3.1 Tests for nutrient deficiency

It is commonly believed that a soil or plant tissue test will show how much nutrient is required by the plant. This is not so. A soil or plant tissue test will only help to identify what is missing or in excess. A soil test will only show that at a certain soil concentration, whether the plant is likely or unlikely to respond to that nutrient. These tests are specific for both soil type and plant being grown (Table 4).

7 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

8 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

Experience suggests that the only worthwhile soil tests will be for P, K, organic matter, soil pH, and soil salt levels. An S test has now been developed. Pulse crops can have different requirements for K, hence different soil test K critical levels.

Table 4: *Adequate levels for various soil test results.*

Nutrient	Test Used		
Phosphorus			
	Colwell	**Olsen**	
Sand	20-30	10-15	
Loam	25-35	12-17	
Clay	35-45	17-23	
Potassium			
	Bicarb.	**Skene**	**Exchangeable K**
Sand	50	50-100	Not applicable
Other soils	100	-	0.25 m.e/100 g
Sandy loam	-	-	-
Faba bean	100-120	-	-
Field pea	70-80	-	-
Lupin	30-40	-	-
Canola	40	-	-
Cereals	30	-	-
Sulfur			
	KCl		
Low	5µg/g (ppm)		
Adequate	8µg/g		

Source: Grain Legume Handbook (2008).

5.4 Soil testing

Key points

- A range of soil test values used to determine if a nutrient is deficient or adequate is termed a critical range.
- Revised critical soil test values and ranges have been established for combinations of nutrients, crops, and soil.
- Nutrient sufficiency is indicated if the test value is above the critical range and therefore there is not likely to be a crop yield response to added nutrients.
- Where the soil test falls below the critical range there is likely to be a crop yield response from added nutrients.
- Soil sampling to greater depth is considered important for more mobile nutrients (N, K, and S) as well as for pH and salinity.
- Use local data and support services to help integrate critical soil test data into profitable fertiliser decisions.

Accurate soil tests allow small landholders to maximise the health of their soils and make sound decisions about fertiliser management to ensure crops and pastures are as productive as possible. Up-to-date critical soil test values will help improve test interpretation to inform better fertiliser decisions. Identifying potential soil limitations enables landholders to develop an action plan (such as an appropriate fertiliser program) to reduce the potential of 'problem' paddocks. [9]

Fertiliser is a major variable cost for grain growers. Crop nutrition is also a major determinant of profit. Both under and over-fertilisation can lead to economic losses due to unrealised potential or wasted inputs.

Before deciding how much fertiliser to apply, it is important to understand the quantities of available nutrients in the soil and where they are located in the soil profile. It is also important to consider whether the fertiliser strategy aims to build, maintain or mine the soil reserves of a particular nutrient. Soil test critical values indicate if the crop is likely to respond to added fertiliser, but these figures do not predict optimum fertiliser rates. Soil test results can be compared against critical nutrient values and ranges, which indicate nutrients that are limiting or adequate. When considered in combination with information about potential yield, last year's nutrient removal and soil type, soil tests can help in making fertiliser decisions.

In the southern region either the Colwell P or DGT P methods are used to assess the availability of P in a soil sample to a crop. It is recommended that a PBI measure accompanies the Colwell P value in order to obtain an indication of the critical Colwell P value as these can vary with different soil types (Table 5). DGT has shown to provide an improve estimate of P availability on calcareous soils and preliminary data also suggests this test may also be useful on acidic soil types with high PBI values. Critical values are freely available from accredited lab providers or from the soil quality factsheet. [10]

Principal reasons for soil testing for nutrition include:

- monitoring soil fertility levels;
- estimating which nutrients are likely to limit yield;
- measuring properties such as pH, sodium (sodicity), and salinity, which affect the availability of nutrients to crops;
- zoning paddocks for variable application rates;
- comparing areas of varying production; and
- as a diagnostic tool, to identify reasons for poor plant performance.

Soil acidity or alkalinity can influence the amount of nutrients available to plants. Table 5 demonstrates nutrient constraints based on soil pH.

9 DAFWA. (2016). Soil sampling and testing on a small property. https://www.agric.wa.gov.au/soil-productivity/soil-sampling-and-testing-small-property

10 GRDC (2016) Monitoring of soil phosphorus, potassium and sulfur. https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/02/Monitoring-of-soil-phosphorus-potassium-and-sulfur

Table 5: *Soil classifications for pH (1:5 soil:water).* [11]

Increasing acidity		Increasing alkalinity
Acidic	Neutral	Alkaline
3 4 5	6 7	8 9 10
Toxicity of :		Toxicity of :
Aluminium (Al)	**Ideal pH Range** for plant growth	Sodium (Na)
Manganese (Mn)		Boron (Bo)
Iron (Fe)		Bicarbonate (HCO3)
Deficiency of:		Deficiency of:
Magnesium (Mg)		Fe
Calcium (Ca)		Zinc (Zn)
Potassium (K)		Mn
Phosphorus (P)		Copper (Cu)
Molybdenum (Mo)		P

5.4.1 Types of test

Appropriate soil tests for measuring soil extractable or plant available nutrients are:

- bicarbonate extractable P (Colwell-P) or DGT for P;
- bicarbonate extractable K (Colwell-K);
- KCl-40 extractable S;
- 2M KCl extractable inorganic N, which provides measurement of nitrate-N and ammonium-N.

For determining crop N requirement, soil testing is unreliable. This is because soil nitrogen availability and crop demand for nitrogen are both highly influenced by seasonal conditions.

Other measurements that aid the interpretation of soil nutrient tests include soil pH, percentage of gravel in the soil, soil carbon/organic matter content, P sorption capacity [currently measured as Phosphorus Buffering Index (PBI)], electrical conductivity, chloride and exchangeable cations (CEC) including aluminium.

Depth for nutrient sampling

The Better Fertiliser Decisions for Cropping (BFDC) project has highlighted that deeper soil sampling provides more appropriate critical soil values and ranges for many soil types. Soil sampling depth for nutrient analysis is currently 0 to 10 centimetres. The 0–10 cm soil layer was originally chosen because nutrients, especially P, and plants roots are concentrated within this layer. Increasingly, there is evidence of the need to assess production constraints, including acidity, in both the surface soil and subsoil layers.

The importance of subsoil K and S contributions to plant nutrient uptake has also been known for a long time. To obtain more comprehensive soil data, including nutrient data, sampling to 30 cm should be considered, providing there are no subsoil constraints (Photo 1). Collecting deeper soil samples does raise issues of logistics and cost, which should be discussed with soil test providers. One suggested approach is to run a comprehensive suite of soil tests on all 0–10 cm samples and only test for N, K, S, and salinity in 10–30 cm samples. On sands, P can also be tested for at depth.

11 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

Note that pH samples need to be taken at 10 cm increments to depth. If sampling to 30 cm, the 0–10 cm, 10–20 cm and 20–30 cm soil layer samples should be tested for pH so that soil acidity can be better understood.

Photo 1: *Nutrients, even relatively immobile ones such as phosphorus (P), can move down the profile in sandy soil, so testing nutrient reserves to depth can be useful.*

Source: GRDC, Photo: Gavin Sarre.

Collecting soil samples for nutrient testing

The greatest source of error in any soil test comes from the soil sample. Detailed sampling instructions are usually provided in soil test kits. The following information is provided as a reference only.

When sampling the 0–10 cm soil layer, 20 to 30 cores per site are required, while for the 10–30 cm soil layer, 8 to 10 cores per site are required. Cores per sample from a uniform zone should be bulked, mixed and sub-sampled for testing. For pH, it is often more useful to see how the figures vary within the paddock or across soil types—therefore, sampling will always be less than ideal. For pH, 8 to 10 cores bulked from six locations in a paddock is usually adequate.

To ensure that a sample is representative:

- check that the soil type and plant growth where the sample is collected are typical of the whole area;
- avoid areas such as stock camps, old fence lines, and headlands;
- ensure that each sub-sample is taken to the full sampling depth;
- do not sample in very wet conditions;
- avoid shortcuts in sampling such as taking only one or two cores, a handful, or a spadeful of soil; and

- avoid contaminating the sample, the sampling equipment and the sample storage bag with fertilisers or other sources of nutrients such as sunscreen, containing zinc.

Critical values and ranges

A soil test critical value is the soil test value required to achieve 90 per cent of crop yield potential. The critical range around the critical value indicates the reliability of the test. The narrower the range, the more reliable the data (Table 6).

Table 6: *Summary table of critical values (mg/kg) and critical ranges for the 0-10 cm sampling layer.*

Soil Test	Crop	Soil Type*	Critical Values (mg/kg)	Critical range (mg/kg)
Colwell-$^{\phi}$	Wheat and barley	Vertosol	17	12-25
		Chromosol/sodosol	22	17-28
		Brown/red chromosol	25	18-35
		Calcarosol	34	26-44
	Barley	Ferrosols	76	46-130
	Canola	All soils	18	16-19
	Field Pea	All soils	24	21-28
Colwell-K	Wheat	Chromosols	40	35-45
		Brown ferrosols	64	57-70
		Kandosols	49	45-52
		Tenosols	41	32-52
	Canola	All soils	45	43-47
	Lupin	Tenosols (WA data)	24	22-27
KCl-40 S+	Wheat	Chromosols/kandosols/ Sodosols/tenosols/ Vertosols	4.5	3.2-6.4
	Canola	NSW data (0 to 15 cm)	8.6	4.8-15.0
		NSW data (0 to 60 cm)	31	25-39

Source: GRDC.

The critical value indicates if a nutrient is likely to limit crop yield based on whether the value is greater than or less than the upper or lower critical range value (Figure 3). If the soil test value is less than the lower limit, the site is likely to respond to an application of the nutrient. For values within the range, there is less certainty about whether a response will occur. In this case, growers have to exercise judgement about the costs and benefits of adding fertiliser in the forthcoming season, versus those associated with not applying. If the soil test is above the critical range, fertiliser is applied only to maintain soil levels or to lower the risk of encountering deficiency. The larger the range around the critical value, the lower the accuracy of the critical value. [12]

12 GRDC. (2014). Crop Nutrition Fact Sheet – Western Region. Soil Testing for crop nutrition. www.grdc.com.au/GRDC-FS-SoilTestingW

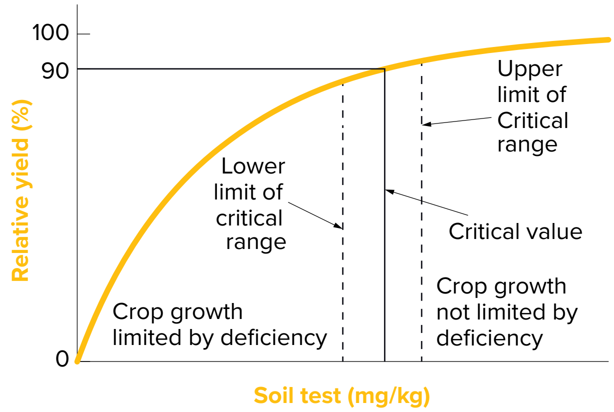

Figure 3: *Generalised soil test response calculation curve. A generalised soil test–crop response relationship defining the relationship between soil test value and per cent grain yield expected. A critical value and critical range are defined from this relationship. The relative yield is the unfertilised yield divided by maximum yield, expressed as a percentage. The BFDC Interrogator fits these curves and estimates critical value and critical range. Normally 90% of maximum yield is used to define the critical value but critical values and ranges at 80% and 95% of maximum yield can also be produced.*

Source: GRDC.

MORE INFORMATION

GRDC Soil testing for crop nutrition – Southern Region Fact sheet.

5.4.2 Southern Australian Soil Quality Program

Key Points

- Soil quality is currently being measured in grain-producing areas across Australia.
- This monitoring program and associated website www.soilquality.org.au provide the Australian grains industry with a unique resource on soil quality including soil biology, chemistry, and physics.
- Each grower's soil quality information is housed on the soil quality website and workshops provide growers with training to access and interpret this information to support improved soil management.

Soilquality.org.au provides an interactive resource to the Australian grains industry on soil quality, including soil biology as well as soil chemistry and physics. The web site allows growers to benchmark their paddocks against values for their local catchment and region as well as against expert opinion. This information aids growers to determine if they are heading in the right direction with their systems and practices, and supports growers to improve soil management practices. The Soil Quality Monitoring Program and the web site www.soilquality.org.au are expanding to include grain producing areas across Australia. This will give growers across Australia access to regionally specific data on soil biological, chemical, and physical constraints to production. This will aid the Australian grains industry to make better management decisions. [13]

5.5 Plant and/or tissue testing for nutrition levels

Plant tissue testing can also be used to diagnose a deficiency or monitor the general health of the pulse crop. Plant tissue testing is most useful for monitoring crop health, because by the time noticeable symptoms appear in a crop the yield potential can be markedly reduced.

13 Soilquality.org. Southern Australian Soil quality program. http://www.soilquality.org.au/factsheets/s-a-soil-quality-program

Why measure nutrients in plant tissues?

Of the many factors affecting crop quality and yield, soil fertility is one of the most important. It is fortunate that producers can manage fertility by measuring the plant's nutritional status. Nutrient status is an unseen factor in plant growth, except when imbalances become so severe that visual symptoms appear on the plant. The only way to know whether a crop is adequately nourished is to have the plant tissue analysed during the growing season.

What plant tissue analysis shows

Plant tissue analysis shows the nutrient status of plants at the time of sampling. This, in turn, shows whether soil nutrient supplies are adequate. In addition, plant tissue analysis will detect unseen deficiencies and may confirm visual symptoms of deficiencies. Toxic levels also may be detected. Though usually used as a diagnostic tool for future correction of nutrient problems, plant tissue analysis from young plants will allow a corrective fertiliser application that same season. A plant tissue analysis can pinpoint the cause, if it is nutritional. A plant analysis is of little value if the plants come from fields that are infested with weeds, insects, and disease organisms; if the plants are stressed for moisture; or if plants have some mechanical injury. The most important use of plant analysis is as a monitoring tool for determining the adequacy of current fertiliser practices. Sampling a crop periodically during the season or once each year provides a record of its nutrient content that can be used through the growing season or from year to year. With soil test information and a plant analysis report, a producer can closely tailor fertiliser practices to specific soil-plant needs.

DO'S

- Sample the correct plant part at the specified time or growth stage.
- Use clean plastic disposable gloves to sample to avoid contamination.
- Sample tissue (e.g. entire leaves) from vigorously growing plants unless otherwise specified in the sampling strategy.
- Take sufficiently large sample quantity (adhere to guidelines for each species provided).
- When troubleshooting, take separate samples from good and poor growth areas.
- Keep samples cool after collection.
- Refrigerate or dry if samples can't be dispatched to the laboratory immediately, to arrive before the weekend.
- Generally sample in the morning, while plants are actively transpiring.

DON'TS

- Avoid spoiled, damaged, dead or dying plant tissue.
- Don't sample plants stressed by environmental conditions.
- Don't sample plants affected by disease, insects or other organisms.
- Don't sample soon after applying fertiliser to the soil or foliage.
- Avoid sample contamination from dust, fertilisers, and chemical sprays, as well as perspiration and sunscreen from hands.
- Avoid atypical areas of the paddock, e.g. poorly drained areas.
- Do not sample plants of different vigour, size, and age.
- Do not sample from different cultivars (varieties) to make one sample.
- Don't collect samples into plastic bags as this will cause the sample to sweat and hasten its decomposition.
- Don't sample in the heat of the day, i.e. when plants are moisture stressed.
- Don't mix leaves of different ages.

Chickpeas should be sampled during the pre-flowering growth stage, with 25–40 samples of the plant collected (Figure 4). [14]

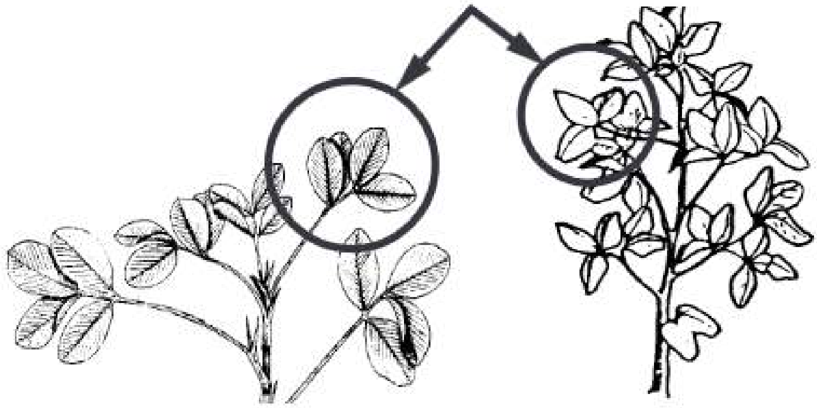

Figure 4: *Collect samples from the whole tops of chickpea plants.*

Source: Spectrum analytic.

Several companies perform plant tissue analysis and derive accurate analytical concentrations; however, it can be difficult to interpret the results and determine a course of action. As with soil tests, different plants have different critical concentrations for a nutrient. In some cases, varieties can differ in their critical concentrations. Table 7 lists the plant analysis criteria for chickpeas. These should be used as a guide only. Care should be taken to use plant tissue tests for the intended purpose.

Table 7: *Critical nutrient levels for chickpea at flowering.*

Nutrient	Plant Part	Critical Range
Nitrogen (%)	Whole shoot	2.3
Phosphorus (%)	Whole shoot	0.24
Potassium (%)	Whole shoot	2.1
Potassium (%)	Youngest mature leaf	1.5
Sulfur (%)	Whole shoot	0.15-0.20
Boron (mg/kg)	Whole shoot	40
Copper (mg/kg)	Whole shoot	3
Zinc (mg/kg)	Whole shoot	12

Most tests diagnose the nutrient status of the plants only at the time they are sampled; they cannot reliably indicate the effect of a particular deficiency on grain yield. Another strategy is to tissue-test a number of paddocks and farms. If there is concern over poor-performing areas, the tissue test can be used to diagnose the potential nutrient deficiency. The critical range (Table 7) can be difficult to use. Wide variations in tissue test results can be due to stress such as frost or waterlogging or even more subtle factors such as solar radiation or time of day of sampling. Although a valuable tool, tissue testing must be used as only one part of an integrated nutrition program. [15]

14 Back Paddock SoilMate. Guidelines for sampling plant tissue for annual cereal, oilseed and grain legume crops. http://www.backpaddock.com.au/assets/Product-Information/Back-Paddock-Sampling-Plant-Tissue-Broadacre-V2.pdf?phpMyAdmin=c59206580c88b2776783fdb796fb36f3

15 Pulse Australia (2013) Northern chickpea best management practices training course manualnd grain legume crops. Limited.

5.6 Fertiliser

Fertiliser recommendations for chickpeas, as with most pulses, tend to be generic, with an over-reliance on the recommendation of MAP-based starter fertilisers across nearly all situations. This is often driven by convenience and availability, rather than meeting the specific nutrient requirements of the crop.

Fertiliser recommendations need to be more prescriptive, and should take into account:

- soil type
- rotation (fallow length and impact arbuscular mycorrhizal fungi (AMF) levels)
- yield potential of the crop
- plant configuration (row spacing, type of opener and risk of 'seed burn')
- soil analysis
- effectiveness of inoculation techniques.

Molybdenum and cobalt (Co) are required for effective nodulation and should be applied as needed. Soil P levels influence the rate of nodule growth. The higher the P level, the greater is the nodule growth. [16]

Nitrogen fertilisers in small amounts (5–15 kg N/ha) are not harmful to nodulation and can be beneficial by extending the early root growth to establish a stronger plant. MAP or DAP fertilisers can be used.

However, excessive amounts of N will restrict nodulation and reduce N fixation.

Inoculated seed and acidic fertilisers should not be sown down the same tube. The acidity of some fertilisers will kill large numbers or rhizobia. Neutralised and alkaline fertilisers can be used.

Acid fertilisers include:

- superphosphates (single, double, triple)
- fertilisers with Cu and/or Zn
- MAP, also known as 11 : 23 : 0 and Starter 12

Neutral fertilisers include:

- 'Super lime'

Alkaline fertilisers include:

- DAP also known as 18 : 20 : 0
- starter NP
- lime[17]

5.6.1 Fertiliser toxicity

All pulses can be affected by fertiliser toxicity. Drilling 10 kg/ha of P with the seed in 18 cm row spacing through 10 cm points rarely caused problems. However, with the changes in sowing techniques to narrow sowing points, minimal soil disturbance, wider row spacing, and increased rates of fertiliser (all of which concentrate the fertiliser near the seed in the seeding furrow), the risk of toxicity is higher.

The effects are also increased in highly acidic soils, in sandy soils, and where moisture conditions at sowing are marginal. Drilling concentrated fertilisers to reduce the product rate per hectare does not reduce the risk.

The use of starter N, e.g. DAP, banded with the seed when sowing pulse crops has the potential to reduce establishment and nodulation if higher rates are used. On sands, up to 10 kg/ha of N at 18 cm row spacing can be safely used. On clay soils, do not exceed 20 kg/ha of N at 18 cm row spacing.

VIDEOS

1. Improving phosphate use efficiency.

16 Lamb, J., & Poddar, A. (1992). Grain legume handbook. South Australian Pea growers Co-operative Ltd., Riverton, South Australia.

17 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

Deep banding of fertiliser is often preferred for lupins— otherwise broadcasting and incorporating, drilling pre-seeding or splitting fertiliser applications so that a lower P rate or no P is in contact with the seed. [18]

5.7 Nitrogen

Key points

- If chickpea plants have effectively nodulated, they should not normally need N fertiliser.
- Nitrate (NO^{3}-) is the highly mobile form of inorganic nitrogen in both the soil and the plant (Figure 5).
- Sandy soils in high rainfall areas are most susceptible to nitrate loss through leaching.
- Soil testing and nitrogen models will help determine seasonal nitrogen requirements. [19]

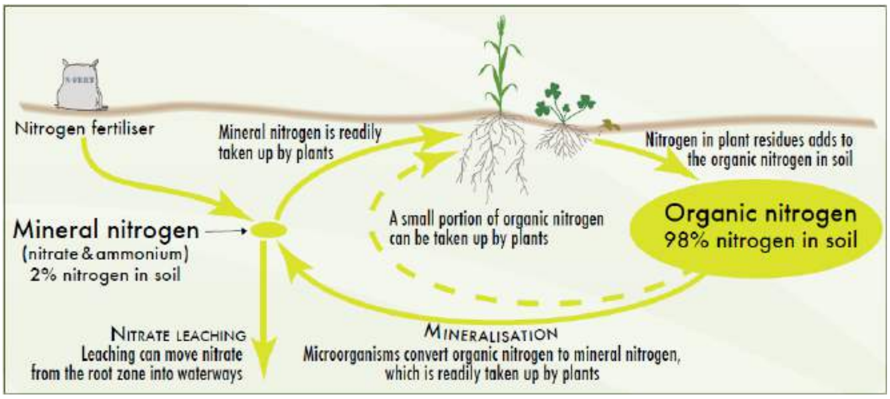

Figure 5: *The soil nitrogen cycle showing the role of mineralisation in making organic nitrogen in soil available for plants to take up.*

Source: Soilquality.org

VIDEOS

2. GCTV14: Nitrogen deficiency.

If chickpea plants have effectively nodulated, they should not normally need N fertiliser (Table 8). Some situations where N fertiliser may warrant consideration include:

- where the grower is unwilling to adopt recommended inoculation procedures
- late or low-fertility planting situations where rapid early growth is critical in achieving adequate height and sufficient biomass to support a reasonable grain yield.

If available soil N is low or sowing is late then "starter" N rates of 5–10 kg/ha may be beneficial. [20] It would be uneconomic to apply N fertiliser rates equivalent to that which would otherwise be fixed by the nodulated chickpea crop.

18 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

19 Soilquality.org. Nitrogen – Western Australia. http://www.soilquality.org.au/factsheets/mineral-nitrogen

20 Pulse Australia. Chickpeas in South Australia and Victoria. http://www.pulseaus.com.au/storage/app/media/crops/2007_Chickpeas-SA-Vic.pdf

Table 8: *Nitrogen balance for chickpeas. Grain harvest index (HI) is the grain yield as a percentage of total shoot dry matter production (average ~40%). Chickpea grain contains 3234 kg N/t.* [21]

Total plant dry matter (t/ha)	Total shoot dry matter yield (t/ha)	Grain yield (t/ha) 40% HI	Total crop N requirement (2.3% N) (kg/ha)	N removal in grain (kg/ha)
1.75	1.25	0.5	40	17
3.50	2.50	1.0	80	33
5.25	3.75	1.5	120	0
7.00	5.00	2.0	160	66
8.75	6.25	2.5	200	83
10.50	7.50	3.0	240	100

IN FOCUS

Effects of below-ground nitrogen on N balances of field-grown faba beans, chickpeas, and barley

The objectives of this study were to quantify below-ground nitrogen (BGN) of rainfed faba beans, chickpeas, and barley and to use the values to determine N balances for the three crops. The BGN fraction of legumes in particular represents a potentially important pool of N that has often been grossly underestimated or ignored in calculating such balances.

The inclusion of BGN in the budgets increased N balances by 38 kg N/ha to +36 kg N/ha for faba beans and by 93 kg N/ha to +94 kg N/ha for chickpea. As there was no external (N_2 fixation) input of N to barley, the inclusion of BGN made no difference to the N balance of the crop of 74 kg N/ha. Such values confirm the importance of BGN of N_2-fixing legumes in the N economies of cropping systems. [22]

Factors influencing nitrogen supply from soils and stubbles

Nitrogen is the key major nutrient influencing crop production in Australian agricultural systems, and maintaining a close balance between inputs and outputs as well as better synchronization between N supply and plant demand is the role of soil management, fertiliser, crop residues, and crops.

Fertiliser N use in Australia increased at an annual rate of approximately 14% compared to that in 1992, which is not only considered economically unsustainable but also environmentally undesirable.

Nitrogen mineralized from soil organic matter and crop residues contributes to a large part of crop N requirements in the rainfed cropping regions across southern Australia. For example, in the year of application, fertiliser N contributes approximately 20–40% of the total N supply of wheat. Soil N supply comes from soil organic matter and recent crop residues and the rate of supply is influenced by the soil biological capacity and modulated by management and environmental factors.

The nitrogen mineralization potential of the top 10 cm soils generally ranges from 10–35 kg N/ha/season in sandy, 25–70 kg N/ha/year in clay and loam soils, and 30–100 kg N/ha/year in red brown earth soils.

The magnitude of soil biological processes and their impact within the farming system varies seasonally due to the variation in the time of their occurrence relative to the

21 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

22 Khan, D. F., Peoples, M. B., Schwenke, G. D., Felton, W. L., Chen, D., & Herridge, D. F. (2003). Effects of below-ground nitrogen on N balances of field-grown fababean, chickpea, and barley. Crop and Pasture Science, 54(4), 333-340.

crop growth and demand. The effect of soil organisms involved in N mineralisation can be seen in both the off-season (fallow) and in-crop season (Figure 6). Nitrogen mineralised during the off-season may accumulate and/or be lost through leaching, denitrification or weed uptake, whereas the N mineralised during the growing season in the rhizosphere may be utilised immediately by the crop. In a farming system, factors influencing nutrient mineralisation–immobilisation processes need to be understood in order to synchronise nutrient availability to plant needs and also to reduce nutrient losses. Additionally, critical periods of biological activity must be taken into consideration to optimise management strategies that help synchronise N supply to availability to crops.

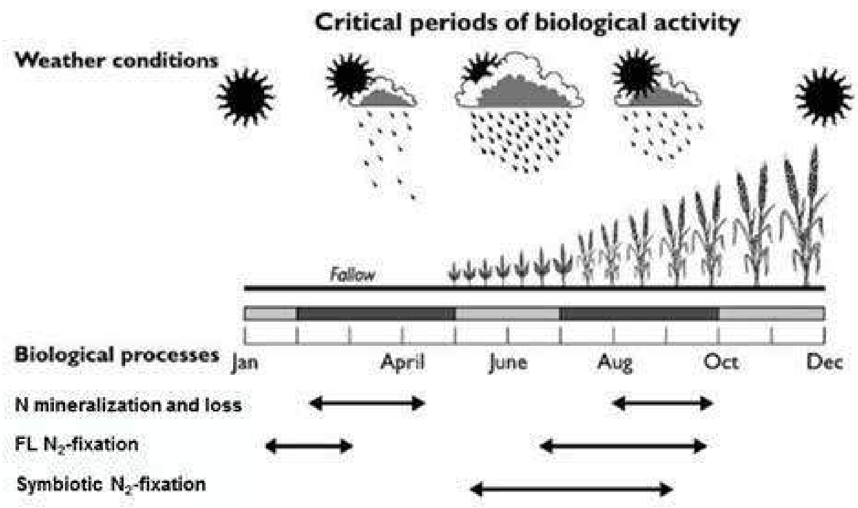

Figure 6: *A conceptual diagram showing functionally important periods for different N-cycling biological processes and their impact within the farming systems in Australian winter-cropping growing regions.*

Source: GRDC.

Decomposition and N mineralization

In low-fertility Australian agricultural soils, crop residues are one of the major sources of carbon (C) for soil biota and retention of stubble after harvest contributes to the conservation of nutrients taken up by the plant within the cropping system. A large portion of N used by crops is mineralised from previous crop and pasture residues through the activity of soil microorganisms (microbial biomass, MB). Decomposition of crop residues is mainly a biological process involving diverse groups of microbial communities and facilitated by the activity of soil fauna. Land use changes from mixed farms where crop rotation with legume pastures was common to continuous cereal cropping generally resulted in a decline in crop residue based N mineralisation. The decline occurred mainly through altered crop residue quality, e.g. wider C:N ratio (100:1) cereal residues replacing N-rich legume residues (15 to 25:1). It is considered that crop residues with a C:N ratio >22:1 generally result in immobilisation (tie-up) of mineral N in microbial biomass. The rate and timing of availability of nutrients from stubble to the following crops is determined by the rate of decomposition and immobilisation (tie-up) by soil microorganisms (N in microbial biomass; MB-N). The amount of N in microbial biomass varies with soil type, crop rotation, tillage, and other management practices that can influence microbial populations (Table 9). In southern Australian cropping regions, the effect of loss of nutrients from stubble removal may be greater than the temporary tie-up of the nutrients during decomposition when retained. However, the scale of these effects vary depending upon stubble load, time and type of burning and tillage.

Table 9: *Amount of nitrogen in microbial biomass and the soil N supply potential as influenced by soil type.*

Location	Soil type	Microbial biomass (kg N/ha)	N supply potential (kg N/ha)**
Waikerie/ Karoonda,SA	Sand and sandy loam	25–45	10–35
Streaky Bay, SA	Calcarosol	30–60	15–50
Kerrabee, NSW	Loam	60–75	35–50
Temora, NSW	Red earth	75–105	50–100
Rutherglen, Vic	Red brown earth	50–100	30–100
Leeton/Warialda, NSW	Clay	50–110	25–75

** N supply potential is calculated from N in MB plus N mineralization measured in a lab-incubation assay.

Source: GRDC.

Stubble retention can provide benefits through changes in soil's physical, chemical, and biological properties. However, the selection of stubble management strategy would have a substantial impact on the potential benefits to be gained from the activity of soil biota in their role in carbon turnover, nutrient mineralisation, and subsequent availability of nutrients to crops. For example, tillage practices accelerate the decomposition and microbial turnover resulting in quick accumulation of mineral N, especially in soils with lower microbial biomass levels. In addition, research from Victoria and South Australia (SA) has shown that tillage can disrupt the linkages between the activity of microbes processing organic N and those related to fertiliser and mineral N transformations influencing the rate of release and accumulation of mineral N in soil. This means strategic tillage practices could be developed to manipulate N release and losses (especially from legume residues), for example to synchronise the release of N to plant demand and avoid losses through leaching and denitrification.

Nitrogen released during decomposition and soil organic matter turnover is rapidly assimilated by MB which is subsequently released through microbial turnover and microbe-fauna interactions. Results from field experiments in SA indicated that in the sandy soils in the Mallee with lower levels of MB, there can be substantial movement (leaching) of mineral N (25–50 kg N/ha; $P < 0.05$) down the soil profile following summer rainfall. Retention of stubble, which generally increases the amount of MB, can therefore arrest the leaching of mineral N to lower depths.

Research at Karoonda in SA, on a dune-swale landscape, has shown that plant type (e.g. wheat, cereal rye, canola, or pasture) can cause large changes in the functional diversity of microorganisms, i.e. microbial communities involved in various biological functions including N cycling processes. Thus, in a crop rotation, such changes coupled with differences in the quantity and quality of organic residues (tops and roots) can significantly modify the N mineralisation-immobilisation processes and availability of N. The magnitudes of these effects vary with soil type and region, which needs to be considered when designing fertiliser N management strategy in a cropping sequence.

Nitrogen fixation—free-living N fixation

Biological nitrogen fixation, by symbiotic and free-living (FL) bacteria can provide economic and environmental sustainability to N management in Australian agriculture. Free living N fixation refers to N fixation by bacteria growing independently in soil or in close association with plant roots where symbiotic N fixation occurs through legume-rhizobia interaction in nodules. Research has shown that communities of free-living and endophytic N-fixing bacteria have been found in association with cereal crops, grasses (including summer active perennial pastures) and non-leguminous plants. With the increased adoption of intensive cropping and area under consecutive cereal crops (>50%), FL-N fixation has the potential to make a major contribution to N requirements in cereal crops.

Additionally, current conservation farming systems support a habitat that promotes activity of FL-N fixing (nifH gene harbouring) bacterial communities both during off-season and in crop, i.e. increased microsites with C availability, wide C/N ratio etc. Improvements in FL-N fixing capacity in soils can provide multiple benefits through reduced requirement for N inputs, disease suppression, C sequestration, etc.

Estimates of FL-N fixation, measured using a laboratory based incubation (15N isotope) assay, ranged from <0.15 to 2.3 kg N fixed/ha/day under optimal soil moisture and temperature conditions. FL-N fixation ranged from 0.2 to 1.5 kg N/ha/day in sand and sandy loam soils in low to medium rainfall regions of southern and Western Australia compared to 0.5 to 2 kg/ha/day in the clay and loam soils in high rainfall regions. The number of optimal days per season does vary in different agricultural regions. The amount of N fixed varied with soil type and influenced by the time of sampling (in crop versus non-crop/fallow period), crop type and mineral nitrogen levels. The amount of FL-N fixed during summer significantly increases (>50%, $P < 0.05$) in the presence of summer-active grasses such as Rhodes grass and Panicum species, compared to winter-cereal crop only systems (Figure 7).

The abundance of FL-N fixing bacteria, percentage clay content (soil type), soil moisture content, and carbon availability are some of the major factors influencing FL-N fixation in cropping soils. Therefore, removal of stubble (one of the major sources of available C) either by burning or grazing would have negative impact on the amount of N fixed by FL-N_2 fixing bacteria. Research in the southern Australian agricultural region has shown that FL-N_2 fixation was higher immediately after harvest and decreased as summer progresses (Figure 7). Thus, careful consideration should be given to how stubble is managed in order to maximise FL-N fixation in cropping soils. Free-living N fixation is generally higher soon after rainfall when the water content is adequate to provide the required low-oxygen conditions (to protect O2-sensitive N fixing enzymes) and carry the carbon to where these bacteria are located. Higher levels of mineral N in the surface soil (0–10 cm) could have a negative effect on the amount of fixation by free-living bacteria, but this varies with soil type so needs region-specific solutions.

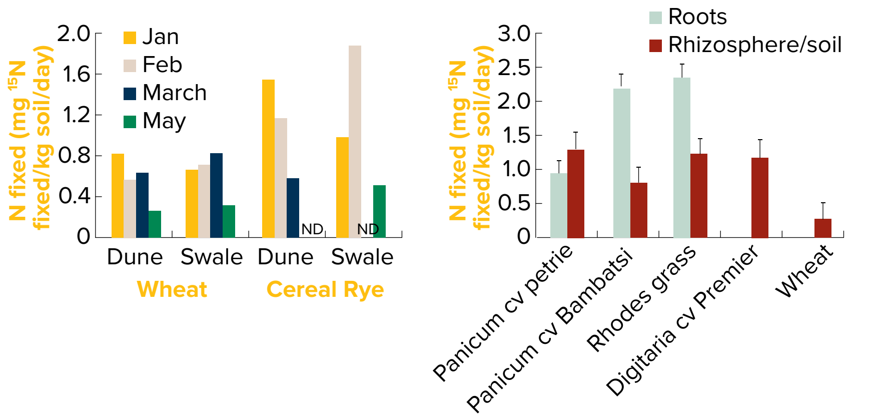

Figure 7: *Amount of free living (FL)-N fixation in soils collected from field experiment at Karoonda in SA during summer of 2011/12 (left) and with summer-active perennial grasses (right).*

Source: GRDC.

Soil type and stubble retention have a large influence on the abundance of nifH-gene harbouring bacteria, for example, abundance increased with clay content ($P < 0.01$) and stubble retention ($P < 0.05$). Populations of FL-N fixing bacteria are generally higher in the rhizosphere soil (soil closely surrounding roots) are generally higher than those found in the bulk soil.

Genetic profiling of N_2-fixing bacteria (nifH gene sequencing analysis) in cereal crop field soils (from SA, QLD, NSW, and WA) indicated the presence of a diverse group of free-living community (112 genera) in different agricultural regions indicating differences based on soil type and environment. Crop and variety types can influence the abundances of various groups thereby affecting the amount of FL-N fixation. Further research could suggest specific management strategies and identify crop varieties that help promote FL-N fixation by specific communities of N_2-fixing bacteria in different soils and regions.

Denitrification and gaseous N losses

The composition and abundance of soil bacteria involved in gaseous N losses (e.g. denitrification and nitrification) varies with soil type, and the denitrification losses are highest where soil nitrate N levels are high and when sufficient biologically available C is present along with low oxygen (O2) concentrations, e.g. water logging. In the southern Australian cropping regions, N losses are sporadic in time and space and vary widely in different agricultural systems. In cropping soils, the primary consideration for reducing gaseous N losses is by matching the supply of mineral N to crop demand and management practices that promote tie-up of N in microbial biomass (immobilisation) generally reduce N losses both through denitrification and leaching.

Nitrification of N fertilisers

The conversion of ammonia and urea N found in commonly used N fertilisers into nitrate N is a biological process mediated by specific group of microorganisms, e.g. nitrifiers, which are mostly abundant in the surface soils. The abundance and the type of nitrifiers present varies with soil type and depth and their activity can be influenced by management practices. Research has shown that banding fertilisers can influence the activity of these microbes and the accumulation of nitrate N. Thus, fertiliser N use efficiency could be manipulated by targeting fertiliser placement or the use of nitrification inhibitors. Immobilisation of fertiliser N in MB, becoming unavailable to plants, is generally short-term and has been found to be available to crops later in the crop season or to the following crop provided it is not leached or lost through gaseous losses.

Conclusions

- Nitrogen mineralised from soil organic matter (SOM) and crop residues makes a large contribution to crop N uptake (>50%).
- A diverse group of microbial communities are involved in the release of nitrate N from SOM and they are present in all agricultural soils.
- Management strategies (such as stubble retention, tillage, fertiliser application, and green manuring) and crop and variety selection can help manipulate microbial communities involved in N mineralisation from organic matter and crop residues and also influence fertiliser N use efficiency.
- Free-living N fixation can make an agronomically important contribution to the available N pool in stubble retained cereal based systems and perennial grass systems. [23]

23 Gupta V. 2016. GRDC Update papers – Factors influencing nitrogen supply from soils and stubbles. https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/02/Factors-influencing-nitrogen-supply-from-soils-and-stubbles

MORE INFORMATION

Factors regulating the contributions of fixed Nitrogen by pasture and crop legumes to different farming systems of eastern Australia.

IN FOCUS

Factors regulating the contributions of fixed nitrogen by pasture and crop legumes to different farming systems of eastern Australia

On-farm and experimental measures of the proportion (%Ndfa) and amounts of N_2 fixed were undertaken for 158 pastures either based on annual legume species (annual medics, clovers or vetch), or lucerne (alfalfa), and 170 winter pulse crops (chickpeas, faba beans, field peas, lentils, lupins) over a 1200 km north-south transect of eastern Australia.

Although pulses often fixed more N than pastures, legume-dominant pastures provided greater net inputs of fixed N, since a much larger fraction of the total plant N was removed when pulses were harvested for grain than was estimated to be removed or lost from grazed pastures.

The net amounts of fixed N remaining after each year of either legume-based pasture or pulse crop were calculated to be sufficient to balance the N removed by at least one subsequent non-legume crop only when below-ground N components were included. This has important implications for the interpretation of the results of previous N_2 fixation studies undertaken in Australia and elsewhere in the world, which have either ignored or underestimated the N present in the nodulated root when evaluating the contributions of fixed N to rotations. [24]

5.7.1 Deficiency symptoms

As proteins make up much of the content of cells, nitrogen is needed in greater quantity that any other mineral nutrient. Nitrogen plays an essential role in the production of chlorophyll, and any deficiency is displayed as yellowing leaves and reduced tillering in cereal crops. This ultimately leads to reduced yields.
Nitrogen is highly mobile within the growing plant, allowing it to re-mobilise and move to tissues that can use it more effectively. As a result, older leaves tend to exhibit nitrogen deficiency symptoms first.

Nitrogen fixation reaches the maximum level at flowering stage and then declines sharply during pod filling. Nitrogen deficiency restricts plant growth and reduce branching. Plants have fewer flowers. Fewer pods are formed resulting in poor yields.

What to look for

1. When nitrogen supply becomes restricted, the older leaves display deficiency symptoms first.
2. The entire plant appears chlorotic, while older leaves turn more yellow than upper leaves (Photo 2).
3. Pink pigmentation develops on the lower part of the stem (Photo 3 left).
4. In prolonged deficiency conditions, the lower leaves turn yellow with reddish-pink margins and a pink colouration develops on the lower stem (Photo 4).
5. In the later stage, the yellow older leaves turn white and drop prematurely (Photo 3 right). [25]

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24 Peoples, M. B., Bowman, A. M., Gault, R. R., Herridge, D. F., McCallum, M. H., McCormick, K. M., ... & Schwenke, G. D. (2001). Factors regulating the contributions of fixed nitrogen by pasture and crop legumes to different farming systems of eastern Australia. Plant and Soil, 228(1), 29-41.

25 Kumar, P., & Sharma, M. K. (Eds.). (2013). Nutrient Deficiencies of Field Crops: Guide to Diagnosis and Management. CABI.

Photo 2: *Nitrogen-deficient crop in foreground compared with nitrogen-fertilised crop behind.*

Source: Kumar, P., & Sharma, M. K. (Eds.). (2013), Photo: Dr P. Kumar.

Photo 3: *Pink pigmentation on lower stem and pale yellow to white chlorotic older leaves (left). Severely deficient whiting yellow leaflets with reddish-pink colouration on the edges (right).*

Source: Kumar, P., & Sharma, M. K. (Eds.). (2013). Photo: Dr P. Kumar.

Photo 4: *Plant showing bottom leaves white, middle leaves yellow, and top leaves green.*

Source: Kumar, P., & Sharma, M. K. (Eds.). (2013). Photo: Dr P. Kumar.

5.7.2 Leaching

Once organic-N is converted to nitrate, it is prone to leaching, particularly in sandy textured soils in high rainfall zones where soil compaction problems slow root growth. Other subsoil constraints, such as soil acidity, may also reduce the efficiency of uptake of NO3- by the crop. Finer textured soils (e.g. red loams) are less likely to suffer significant NO3- to leaching, allowing efficient use of available nitrogen.

5.7.3 Managing Nitrogen

Cropping systems using carefully designed species mixtures may be a way to lower N fertilisation input, while maintaining economic profitability. [26]

Nitrogen fertilisers in small amounts (5–15 kg N/ha) are not harmful to nodulation and can promote early root growth to establish stronger plants. Fertiliser compounds such as MAP and DAP are suitable for chickpea production. Excessive amounts of nitrogen however, will restrict nodulation and reduce nitrogen fixation.

'Starter N' may be beneficial, but is not essential. [27]

Optimising nitrogen fixation in southern farming systems

Pulse and pasture legumes can provide an abundant, inexpensive, and sustainable source of nitrogen (N) for Australian cropping systems.

26 Hirel, B., Tétu, T., Lea, P. J., & Dubois, F. (2011). Improving nitrogen use efficiency in crops for sustainable agriculture. Sustainability, 3(9), 1452-1485.

27 Pulses Australia. Chickpea Production: Southern and Western Region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

Research at the South Australian Research and Development Institute (SARDI) is aiming to optimise N_2-fixation in southern farming systems by: improving our understanding of how cultivars differ in their N_2-fixation potential; clarifying where legumes respond to inoculation; identifying key agronomic factors affecting fixation; and testing new inoculants. The work has so far mainly focused on field peas.

Optimising agronomy

Agronomic practices such as time of sowing, crop nutrition, and managing disease and weed pressures—which optimise dry matter production of the pulse crop—also drive nitrogen demand and generally encourage N_2-fixation. Other factors can have more direct impacts on the symbiosis.

Mineral N at sowing

Pre-sowing mineral N in the soil (and also N applied in fertiliser at sowing) can impact on N_2-fixation. High levels of mineral N in the top 10 cm of soil at sowing (>30 kg/ha) may reduce the number of nodules which form per plant and will reduce N_2-fixation. When 30 kg of N was added at sowing, nodulation was reduced by 10 per cent and N_2-fixation by an average of 10 kg/ha.

Herbicides

Plant back times for herbicides should be strictly adhered to. Residues from sulfonylurea (SU) herbicides are known to retard root growth and development and the ability of roots to form nodules and then fix nitrogen. In-crop herbicides which cause significant yellowing and temporary stunting of crops also have the potential to reduce nitrogen fixation of the crop. These effects are thought to be more pronounced on light textured soils and when multiple stress factors are present (e.g. water, frost, SU residues).

Contributions of fixed N from legume roots are presently not well quantified and some work has begun to gain a better understanding of these contributions and how they might be managed. [28]

MORE INFORMATION

Soil Nitrogen supply factsheet.

5.8 Phosphorus

Key points

- Chickpeas are not as responsive to phosphorus fertiliser as some of the other pulses. In order to match the nutrient requirement of a crop yielding 1.5–3.5 t/ha, a guide for alkaline soils with a good fertiliser history is 7–16 kg/ha of phosphorus. This is equivalent to 80–186 kg/ha of single super or 40–95 kg/ha of double super. [29]
- Phosphorus cycling in soils is particularly complex, and agronomic advice is recommended when interpreting soil test results.
- Only 5–30% of phosphorus applied as fertiliser is taken up by the plant in the year of application.
- Phosphorus does not move readily in soils, except very light sandy soils in high rainfall areas.

Ancient and highly weathered soils with very low levels of natural phosphorus (P) dominate much of Australia. Many of our agricultural soils are among the most acutely phosphorus-deficient in the world, and profitable crop production has only been possible through significant applications of P-fertilisers.
Phosphorus is an essential element for plant and animal growth and important during cell division and growth.
Complex soil process influence the availability of phosphorus applied to the soil, with many soils able to 'tie up' phosphorus, making it unavailable to plants. The

28 E. Farquharson, N. Charman and R. Ballard (SARDI). 2016. Optimising nitrogen fixation in southern farming systems. https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/02/Optimising-nitrogen-fixation-in-southern-farming-systems

29 Pulse Australia. Chickpeas in South Australia and Victoria. http://www.pulseaus.com.au/storage/app/media/crops/2007_Chickpeas-SA-Vic.pdf

soil's ability to do this must be measured when determining requirements for crops and pastures. [30]

Soil phosphorus levels influence the rate of nodule growth. The higher the phosphorus level the greater the nodule growth. A 2 t/ha chickpea crop will on average remove approximately 6.5 kg/ha of phosphorus. This then is the minimum amount of phosphorus that needs to be replaced. Higher quantities may be needed to build up soil fertility or overcome soil fixation of phosphorus. [31]

Chickpeas are adapted to alkaline soils with high levels of unavailable P, and have evolved methods of extracting P, and some other nutrients, from the soil that would be inaccessible to many other pulse and cereal crops.

This ability is largely due to a combination of two factors: organic acids secreted from the root system and arbuscular mycorrhizalfungi (AMF) colonising the chickpea root system increasing uptake of P and Zn. More P may be required in low AMF situations (e.g. after a long fallow). High rates of (P) and Zinc will be required in most long fallow situations (fallows longer than 10 months) where soil VAM levels may be low. [32]

Based on information from the Northern region, chickpea is considered dependent on AMF to reach yield potential so yield reduction of 60–80% can occur in low AMF situations. [33]

High AMF situations

Where soil AMF levels are moderate–high (double-crop situations or short, six-month fallows from wheat), consistent responses to applied phosphate fertiliser are only likely where soil bicarbonate-P levels fall <6 mg/kg and are critically low.

Low AMF situations

Levels of AMF become depleted as fallow length is increased (Table 10), or after crops such as canola that do not host AMF growth. In these conditions of low AMF (long fallows of >8–12 months), chickpeas are very responsive to applied P and Zn. Although chickpeas in this situation will usually show a marked growth response to starter fertilisers (Table 11), this may not always translate into a positive yield response.

The most cost-effective strategy in a long fallow situation (low AMF) may be to ensure that the paddock is sown relatively early in the recommended sowing window, so that sufficient time is allowed for the crop to recover from the delay in early growth. These recommendations are based on soil samples taken to a depth of 0–10 cm.

Table 10: *An example of effect of fallow length on arbuscular mycorrhizae (AM) spore survival, and crop yield response to fertilisation after the fallow.*

Fallow duration (months)	AM Spores (no./g soil)	Crop yield (kg/ha)	
		Nil (P & Zn)	+ (P & Zn)
21	14	2865	4937
11	26	3625	3632
6	44	5162	4704

Source: J Thompson (1984).

Results in Table 11 show that chickpea growth on short-fallow land (six months after wheat) was much better than growth after long fallow on the same property. The addition of P and Zn fertilisers could not entirely compensate for the lack of AMF in chickpea on the long fallow.

30 Soilquality.org. Phosphorus – Western Australia. http://www.soilquality.org.au/factsheets/phosphorus

31 Pulses Australia. Chickpea Production: Southern and Western Region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

32 L Jenkins, K Moore, G Cumming. Pulse Australia. Chickpea: High Quality seed. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/high-quality-seed

33 Pulses Australia. Chickpea Production: Southern and Western Region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

MORE INFORMATION

VAM and long fallow disorder.

VIDEOS

3. CTV13: Phosphorus deficiency.

Table 11: *Effect of fallow length and fertiliser on chickpea growth (based on Northern region research).* [34]

Fallow duration (months)	Dry weight (g/plant) of chickpea at 12 weeks			
	Nil fertiliser	P (50 kg/ha)	Zn (10 kg/ha)	P & Zn
Long (14 months)	1	1.2	0.4	1.9
Short (six months after wheat)	3.1	2.8	2.7	3.3

One study found that there is a poor relationship between the commonly used indicator (Colwell P 0–10 cm) and the response to added P to chickpea. It has been suggested that a more reliable test than the Colwell P determination is warranted to get more efficient use out of applied P to inherently low P soils. [35]

5.8.1 Deficiency symptoms

Phosphorus deficiency is difficult to detect visually in many field crops, as the whole plant tends to be affected. Stunted growth, leaf distortion, chlorotic areas, and delayed maturity are all indicators of phosphorus deficiency. Phosphorus is concentrated at the growth tip, resulting in deficient areas visible first on lower parts of the plant.

A purple or reddish colour associated with accumulation of sugars is often seen in deficient plants, especially when temperatures are low. Deficient crops are often poorly tillered. Visual symptoms, other than stunted growth and reduced yield, are not as clear as are those for nitrogen and potassium. At some growth stages, phosphorus deficiency may cause the crop to look darker green.

The role of phosphorus in cell division and expansion means crop establishment and early growth is highly dependent on sufficient sources of the nutrient. Trials have shown significant agronomic penalties from applying phosphorus more than 10 days after germination. Most of these phosphorus timing trials indicate the optimum time for P-fertiliser application is before or during seeding. [36]

What to look for

1. Affected stems develop a reddish purple pigmentation that intensifies and becomes darker in prolonged deficiency conditions. (Photo 5).
2. In phosphorus-deficient plants, the top edges and upper surface of the leaflets exhibit reddish-purple discolouration (Photo 6). [37]

34 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

35 Routley, R., Spackman, G., & Conway, M. (2008). Variable response to phosphorous fertilisers in wheat and chickpea crops in central Queensland.

36 Soilquality.org. Phosphorus – Western Australia. http://www.soilquality.org.au/factsheets/phosphorus

37 Kumar, P., & Sharma, M. K. (Eds.). (2013). Nutrient Deficiencies of Field Crops: Guide to Diagnosis and Management. CABI.

Photo 5: *Plant showing dark green leaves with reddish-purple discolouration in older leaves.*

Source: Kumar, P., & Sharma, M. K. (Eds.). (2013). Photo: Dr P. Kumar.

Photo 6: *Purpling appearing on edges of leaflets (left) through to purple pigmentation spreading inwards to cover upper srface of leaflets (right).*

Source: Kumar, P., & Sharma, M. K. (Eds.). (2013). Photo: Dr P. Kumar.

5.8.2 Do late sown crops need extra phosphorus?

As winter sowing dates get later, a crop's potential response to Phosphorus (P) fertiliser rises at the same time absolute yield potential is falling. This raises the question of what adjustments might need to be made to P fertiliser rates.

Plant physiology indicates that **later sown crops should be more responsive to P fertiliser**. Later sown winter crops usually grow more slowly. Their smaller root systems may reduce P uptake from the soil, compared to an early sown crop with a more developed root system, in contact with a greater volume of soil. **Actual**

trial results suggest response to higher rates of P fertiliser at late sowing is uncertain. [38]

5.8.3 Fate of applied fertiliser

Phosphorus fertiliser is mostly applied in a water soluble form which can be taken up by plants, retained by soil and lost through erosion and leaching (Figure 8). In the water soluble form phosphorus is not stable, and rapidly reacts in the soil (principally with iron, aluminium, and calcium) to form insoluble, more stable compounds. Therefore, competition between the soil and plant roots for water soluble phosphorus arises, with only 5% to 30% of the phosphorus applied taken up by the crop in the year following application. Furthermore, at low pH (<5.0), the soil's ability to fix phosphorus rises dramatically, thereby decreasing plant availability. [39]

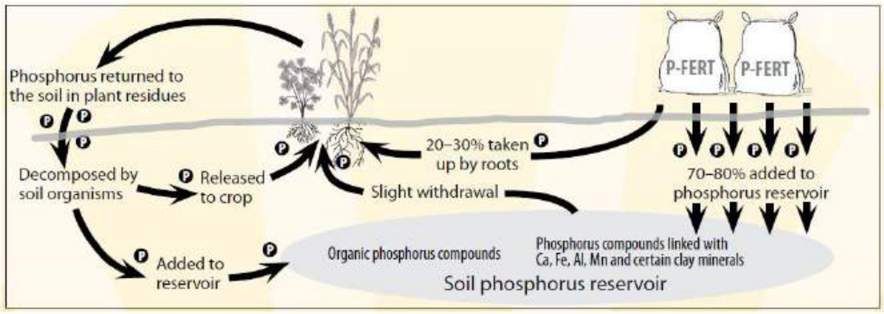

Figure 8: *The phosphorus cycle in a typical cropping system is particularly complex, where movement through the soil is minimal and availability to crops is severely limited*

from Glendinning, 2000, in Soilquality.org.

5.8.4 Measuring a soil's ability to fix phosphorus

Knowing the soil's ability to fix phosphorus is vital in determining the rates of fertiliser application. A high-fixing soil will require significantly more P-fertiliser, and commercial tests have been developed to determine this. These are used in conjunction with other soil and crop traits to optimise fertiliser P requirements:

1. **Reactive Iron Test** measures the amount of iron extracted from soil by ammonium oxalate. This indirect measure of a soil's ability to fix P is only accurate when soil is adjusted for pH.
2. **Phosphorus Retention Index** (PRI) is a direct measure of P-sorption and involves mixing a quantity of soil in solution with a single amount of P for a set period of time. The amount of P remaining in solution measures the soil's ability to fix phosphorus.
3. **Phosphorus Buffering Index** (PBI) is similar to PRI except that a range of P rates are mixed with the soil, and the index is adjusted for pH. This is becoming the Australian standard for measuring soil P-sorption.
4. **Diffuse Gradient Technology Phosphorus** (DGT-P) is a relatively new method currently being tested for use with Australian soils, and mimics the action of the plant roots in accessing available phosphorus (see DGT-P factsheet). [40]

5.8.5 Phosphorus retention and removal

Phosphorus that is not removed from the soil system remains as (i) undissolved in fertiliser granules, (ii) adsorbed by the soil, or (iii) present in organic matter. These sources all supply some P for plant uptake and thus maintain a residual fertiliser value. A long term regime of applying P fertiliser decreases the capacity of the soil to

38 Conyers, M. 2016. Do late crops need extra phosphorus? https://extensionhub.com.au/web/crop-nutrition/-/do-late-sown-crops-need-extra-phosphoru-1

39 Soilquality.org. Phosphorus – Western Australia. http://www.soilquality.org.au/factsheets/phosphorus

40 Soilquality.org. Phosphorus – Western Australia. http://www.soilquality.org.au/factsheets/phosphorus

adsorb phosphorus, giving increased effectiveness of subsequent applications. Each crop species will remove different amounts of phosphorus from soil following harvest (see Table 1), and must be accounted for during nutrient budgeting.

5.8.6 Leaching and placement of phosphorus

Phosphorus movement in soil varies depending on soil type, although it generally stays very close to where it is placed. With the exception of deep sandy soils, very little phosphorus is lost to leaching. Tests on loamy and clay soils with a history of P-fertiliser application show a rapid reduction in phosphorus with depth.

Agronomic benefits of banding P-fertiliser on high fixing soils have only been evident in trials with lupins, with this attributed to less soil coming in contact with the concentrated phosphorus layer. Wheat and canola have not responded to banded phosphorus on high fixing soils.

Placing high rates of phosphorus close to germinating seedlings can reduce germination and establishment, and should be placed at least 2 cm below the seed. Some considerations when banding phosphorus are:

- Drying conditions in the furrow following seeding, where a "salting" effect draws moisture from around the seed.
- Canola and lupins are more sensitive to higher phosphorus concentrations.
- Higher concentration of fertiliser in furrow when seeding at higher row spacing.
- Nitrogen-containing fertilisers (e.g. DAP) are more damaging than superphosphate fertilisers. [41]

5.8.7 Soil P testing

The Soil P test needs to be interpreted in association with the soil's P-adsorption capacity, which is estimated by the PBI. The higher the PBI value, the more difficult it is for a plant to access P. Phosphorus is relatively immobile in soils and P applied to the 0 to 10 cm layer tends to remain in that layer, especially in no-till systems. This is the case for loams, duplexes, and red and yellow sands. However, grey sands have low P sorption capacity and P can leach from the 0 to 10 cm soil layer and accumulate in the layers below 10 cm. [42]

5.9 Sulfur

Sulfur (S) is needed at higher rates for chickpea. Sulfur is an important nutrient in the production of proteins and as such is used at a higher proportion than other non legume crops. On a relative yield basis S supply is similar for all crops, except canola which has a higher demand.

Use "grain legume" fertilisers. If the paddock has a history of single super and/or gypsum use, then S may be adequate, particularly on clay soils. Prolonged use of double or triple super could lead to an S deficiency, especially on lighter soils.

Historically, S has been adequate for crop growth because S was supplied in superphosphate. Sulfur deficiency occurs when growers repeatedly use high analysis N and P fertilisers that are low in S and in wet growing seasons due to leaching of S. Occurrence of S deficiency appears to be a complex interaction between the seasonal conditions, crop species, and plant availability of subsoil S. As with N, these factors impact on the ability of the soil S test to predict plant available S. [43]

Certain soil types are prone to S deficiency—for example, some basaltic, black earths. On these soils with marginal S levels, deficiency is most likely to occur with double-cropping where levels of available S have become depleted, for example.

41 Soilquality.org. Phosphorus – Western Australia. http://www.soilquality.org.au/factsheets/phosphorus

42 GRDC. (2014). Crop Nutrition Fact Sheet – Western Region. Soil Testing for crop nutrition. www.grdc.com.au/GRDC-FS-SoilTestingW

43 GRDC. (2014). Crop Nutrition Fact Sheet – Western Region. Soil Testing for crop nutrition. www.grdc.com.au/GRDC-FS-SoilTestingW

5.9.1 Symptoms

- Sulfur deficiency symptoms are often seen in the early growth stage of the crop.
- Sulfur deficient plants become smaller and slender.
- The yield is severely reduced as the deficient plants produce fewer pods and smaller seeds.
- Deficiency symptoms of sulfur first appear and become more severe in younger leaves (Photo 7, left).
- Younger leaves turn pale green to pale yellow, while the lower leaves remain dark green.
- In severe deficiency conditions, the youngest leaflets turn completely yellow (Photo 7, right) and the entire plant can turn chlorotic. [44]

Photo 7: *Yellowing intensified on younger leaflets (left). Leaflets showing uniform yellowing (right).*

Source: Kumar, P., & Sharma, M. K. (Eds.). (2013). Photo: Dr P. Kumar.

5.9.2 Applying sulfur

Application of 5–10 kg S/ha will normally correct S deficiency. Where soil phosphate levels are adequate, low rates of gypsum are the most cost-effective, long-term method of correcting S deficiency.

Granulated sulfate of ammonia is another effective option where low rates of N are also required.

Marked responses to 25 kg/ha of sulfate of ammonia have been observed when sowing chickpeas in double-crop situations. [45]

IN FOCUS

Growth, nitrogen fixation and nutrient uptake by chickpea (*Cicer arietinum*) in response to phosphorus and sulfur application under rainfed conditions in Pakistan.

A field experiment was conducted to assess the seed yield, nitrogen fixation and nutrient uptake by chickpea (Cicer arietinum L.) in response to application of different levels of phosphorus (P) and sulfur (S). The treatments comprised three levels (0, 40, and 80 kg P 2O 5 ha -1) of P and three levels (0, 15, and 30 kg S ha -1) of S from two sulfur sources (gypsum & ammonium sulfate) in different combinations. In a soil with 3 ppm of phosphorus and 6 ppm of sulfur, application of P and S resulted in significant yield increases under rainfed conditions. The addition of sulfur had a direct effect on N fixation and also resulted in the improvement of

44 Kumar, P., & Sharma, M. K. (Eds.). (2013). Nutrient Deficiencies of Field Crops: Guide to Diagnosis and Management. CABI.

45 Pulse Australia (2013) Northern chickpea best management practices training course manual http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/northern-guide Limited.

protein content. Application of P and S resulted in significant increase in seed yield by 21% and 12% more than control, respectively. Sulfur application had significant effect on percent nitrogen derived from atmosphere (% N dfa), while effect of P was non-significant. There was significant increase in protein content of chickpea seed due to application of S. Application of both P and S resulted in increase in nitrogen (N) fixation by 16%. An economic analysis indicated that the most profitable application of P and S on this soil was 40 kg/ha P and 30 kg/ha S. [46]

5.10 Potassium

Diagnosis of potassium (K) deficiency before visual symptoms occur is important in order to avoid large yield losses. K is mobile and readily transferred from old to young leaves when a deficiency occurs.

Factors such as soil acidity, soil compaction, and waterlogging will modify root growth and the ability of crops to extract subsoil K. Consequently, interrogation of results across all soil types has identified a poor relationship between the soil test for K and crop yield response.

However, the critical value (0 to 10 cm) for K is defined across all soil types as 41 mg K/kg to achieve a relative yield of 90% (for wheat). [47]

IN FOCUS

Diagnosis of potassium deficiency in faba bean and chickpea by plant analysis

Critical potassium (K) concentrations for the diagnosis of K deficiency were determined in various shoot parts of faba beans (Vicia faba L.cv. Fiord) and chickpeas (Cicer arietinum L. T1587) plants grown at K rates of 0–240 mg K/kg in a K-deficient soil in the glasshouse. It is recommended that the critical values for the diagnosis of K deficiency at 7–8-leaf stages are 1.3–1.5% in the youngest fully extended leaf (YFEL), 1.1–1.2% in the 1st plus 2nd leaf blades below the YFEL and 1.8–2.0% in whole shoot of faba bean, and 1.4–1.5% in YFEL, 2.7–2.8% in the 1st plus 2nd leaf petioles, and 2.1–2.2% in whole shoot of chickpea. [48]

46 Islam, M., Mohsan, S., Ali, S., Khalid, R., UL-HASSAN, F. A. Y. Y. A. Z., Mahmood, A., & Subhani, A. (2011). Growth, Nitrogen Fixation and Nutrient Uptake by Chickpea (*Cicer arietinum*) in Response to Phosphorus and Sulfur Application under Rainfed Conditions in Pakistan. International Journal of Agriculture & Biology, 13(5).

47 GRDC. (2014). Crop Nutrition Fact Sheet – Western Region. Soil Testing for crop nutrition. www.grdc.com.au/GRDC-FS-SoilTestingW

48 Aini, N., & Tang, C. (1998). Diagnosis of potassium deficiency in faba bean and chickpea by plant analysis. Animal Production Science, 38(5), 503-509.

5.10.1 Symptoms

Photo 8: *Tips of leaflets show brown necrotic patches and eventually die.*

Photos: Michael Bell, QAAFI.

Photo 9: *Margins and tips of lower leaves show chlorosis.*

Photos: Michael Bell, QAAFI.

5.10.2 Applying potassium

Responses to K are unlikely on most black earths and grey clays. Potassium fertilisers may be warranted on red earths (kraznozems) but this should be based on soil analysis. Fertiliser responses are likely where soil test levels using the ammonium acetate test fall below:

- exchangeable K of 0.25 meq/100 g (or cmol/kg) on black earths and grey clays
- exchangeable K of 0.40 meq/100 g K on red earths and sandy soils.

Application of 20–40 kg K/ha banded 5 cm to the side of, and below, the seed line is recommended in situations where soil test levels are critically low. Alternatively, blends such as Crop King 55 (13 N,13 P,13 K) may be considered at rates of 80–120 kg/ha in situations where K levels are marginal. [49]

5.11 Micronutrients

Why is there a need for micronutrients/trace elements?

Essential trace elements are nutrients which are required by plants and animals to survive, grow, and reproduce but are needed in only minute amounts. Southern cropping soils are more likely to be deficient in zinc (Zn), copper (Cu), and manganese (Mn) than the other trace elements.

Of these three, Zn deficiency is probably the most important because it occurs over the widest area. Zn deficiency can severely limit annual pasture legume production and reduce cereal grain yields by up to 30 per cent.

Cu deficiency is also important in pollen viability, carbohydrate and protein synthesis, photosynthesis, cell wall stability and other metabolic functions. Deficiency occurs in limited soils, and needs to be monitored by soil and plant tests, local knowledge of responses, and in field trials/strips. Cu deficiency can cause significant losses.

If these three trace elements are not managed well the productivity of crops and pastures can suffer economic losses, and further production can also be lost through secondary effects such as increased disease damage and susceptibility to frost.

Adequate trace element nutrition is just as important for vigorous and profitable crops and pastures as adequate major element (such as nitrogen or phosphorus) nutrition. [50]

Molybdenum and cobalt are required for effective nodulation and should be applied as needed. Foliar sprays of zinc and manganese are a useful method of treating deficiency on high pH soils where soil fixation of these nutrients occur. [51]

5.11.1 Zinc

Zinc occurs in low levels in all Australian soils and is known to be deficient in many of the areas that chickpeas are grown, including the sandy and calcareous soils of Northern Vic and SA. As an important nutrient to all crop growth and understanding of zinc nutrition is vital to the successful chickpea production.

Soils low in available zinc (Zn) occur in many areas of the world where chickpea is grown. Improving the ability to grow and produce high yield under limited supplies of Zn (often referred to as Zn efficiency) may increase productivity of chickpea in many of these regions.

Chickpea is considered to have a relatively high demand for zinc, but also possess highly efficient mechanisms for extracting Zn from the soil. Zinc seed treatments may be a cost-effective option in situations where soil P levels are adequate but zinc levels are likely to be deficient.

Chickpeas are prone to zinc (Zn) deficiency. Low or marginal zinc levels are widespread in many cropping districts. Zinc, and to a lesser extent iron, deficiency is prevalent on calcareous soils, particularly dark brown clay soils with high pH.

Zinc applications lasts about two years on calcareous clays and 6–7 years on loamy soils. Zinc is not mobile in the soil and an even distribution is important. Zinc

49 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

50 GRDC Update Papers. 2016. Detecting and managing trace element deficiencies in crops. https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/02/Detecting-and-managing-trace-element-deficiencies-in-crops

51 Matthew Witney (2016). Personal Communication.

can be applied by spray to the soil, in furrow, coated on granular fertiliser or as a foliar spray. [52]

Zinc deficiency affects plant-water relationships, induces stomatal closure and decreases transpiration in plants.

IN FOCUS

Response of chickpea genotypes to zinc fertilisation under field conditions in South Australia

The effects of Zn on the growth, grain yield and tissue ZN concentration of a number of chickpea genotypes were compared in one field experiment in South Australia. The DPTA-extractable Zn at the sites ranged from 0.24 to 0.30 mg kg–1. In each experiment 10 genotypes were grown with or without additional Zn. Except for Tyson, the genotypes differed between the two experiments in South Australia. Grain yield responses to applied Zn ranging from 7% to 19%, occurred at each site. The rankings for Zn efficiency from the field experiments were significantly correlated with the rankings in previous pot trials. The high levels of zinc efficiency suggested that significant genetic gains in productivity under conditions of low Zn supply are possible. The ability of pot trials to predict performance under field conditions indicates that screening for zinc efficiency can be successfully conducted in the glasshouse. [53]

Symptoms

It is very difficult to diagnose Zn deficiency in pasture or grain legumes because the characteristic Zn deficient leaf markings are rarely produced in the field. Zn deficiency causes shortening of stems and the leaves fail to expand fully. This results in plants which appear healthy but are stunted and have small leaves. Plant symptoms appear to be worst early in the season when conditions are cold and wet and light intensity is low. In spring, symptoms often do not appear on new leaves but grain yields will usually be reduced.

- Zinc deficient plants appear stunted and have fewer branches. The size of leaflets is reduced. Crop maturity gets delayed.
- The younger leaves become pale green first, then a reddish-brown discolouration appears on margins of leaflets and on the lower parts of the stem (Photo 10, left).
- In severe deficiency, bronzing and necrosis occurs on the leaflets (Photo 10, right). [54]

52 Pulses Australia. Chickpea Production: Southern and Western Region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

53 Khan, H. R., McDonald, G. K., & Rengel, Z. (2000). Response of chickpea genotypes to zinc fertilisation under field conditions in South Australia and Pakistan. Journal of plant nutrition, 23(10), 1517-1531.

54 Kumar, P., & Sharma, M. K. (Eds.). (2013). Nutrient Deficiencies of Field Crops: Guide to Diagnosis and Management. CABI.

Photo 10: *Reddish-brown pigmentation spreading on the entire upper surface of the leaflets (left). Deficient leaflets showing reddish pigmentation and necrosis on the margins (right).*

Source: Kumar, P., & Sharma, M. K. (Eds.). (2013). Photo: Dr P. Kumar.

Correcting Zinc deficiency in southern Australia

Correction of Zn deficiency in a way which provides benefits after the year of treatment is possible through the use of Zn-enriched fertilisers or a pre-sowing spray of Zn onto the soil (incorporated with subsequent cultivations).

Another option that will also provide long term benefits but has become available only recently is the application of fluid zinc at seeding. The advantage of this approach is that it will provide residual benefits for subsequent crops and pastures and has a low up-front application cost (providing you ignore the capital investment in a fluid delivery system!). At current prices, a typical application may cost about $6.00/ha (this is 1 kg of Zn/ha).

Only Zn-enriched fertilisers of the homogenous type (fertiliser manufactured so that all granules contain some Zn) are effective at correcting Zn deficiency in the first year of application. A rate of two kilograms of elemental Zn per hectare applied to the soil is necessary to overcome a severe Zn deficiency and should persist for three to ten years (depending on soil type). Short intervals between repeat applications of Zn will be necessary on heavy and calcareous soils in the high rainfall areas, while seven to ten year intervals will be acceptable in the low rainfall areas. Following an initial soil application of 2 kg Zn/ha repeat applications of 1 kg/ha will probably be sufficient to avoid the reappearance of Zn deficiency in crops and pastures. Most zinc-enriched fertilisers are now not sold as pure homogeneous types, but providing a homogeneous fertiliser is used as part of the mix then the final product is still satisfactory for correcting Zn deficiency. For example, the company may produce a diammonium phosphate (DAP) Zn five per cent 'parent' product which has Zn on every granule which they will then blend with straight DAP to give 1 and 2.5 per cent products for the retail market.

Zn deficiency can be corrected in the year that it is recognised with a foliar spray of 250–350 g Zn/ha but it has no residual benefits and is therefore not the best approach for a long-term solution. Zinc can be mixed with many herbicides and pesticides but not all, so check with your supplier for compatible tank mixes before you make the brew. Recent trials in eastern Australia suggest that chelated sources of trace elements are no more effective at correcting a deficiency than sulfates (see Photo 1 for an example of treating copper deficiency in wheat), although older results from WA showed that there are situations where they can be superior.

Seed dressings of zinc are another option for managing Zn deficiency. These products are effective and will supply Zn to the young crop but they will not completely overcome a severe deficiency. Nor will they increase soil reserves of Zn. Seed with high internal levels of Zn can also be used in a similar way. However, both approaches should be used in conjunction with soil applications to correct and manage Zn deficiency in the long term. This option will currently cost approximately $3.00/ha. [55]

Applying Zinc

There is a lack of Australian and overseas research on Zn responses in chickpeas, and Zn fertiliser recommendations are being conservatively based on a general recommendation used for all crops. Based on DTPA analysis of soil samples at 0–10 cm, critical values of Zn are:

- below 0.8 mg/kg on alkaline soils
- below 0.3 mg/kg on acid soils.

In the Northern region, AMF are important to Zn nutrition in chickpeas, and large responses can be expected where AMF levels have become depleted due to long fallows (over 8–10 months). This is not so much of an issue in the Southern region.

Pre-plant treatments

Severe Zn deficiency can be corrected for a period of 5–8 years with a soil application of 15–20 kg/ha of zinc sulfate monohydrate, worked into the soil 3–4 months before sowing.

Zinc is not mobile in the soil and needs to be evenly distributed over the soil surface, and then thoroughly cultivated into the topsoil. In the first year after application, the soil-applied Zn may be not fully effective and a foliar Zn spray may be required.

Fertilisers applied at sowing

A range of phosphate-based fertilisers either contain, or can be blended with, a Zn additive.

Foliar zinc sprays

A rate of two kilograms of elemental Zn per hectare applied to the soil is necessary to overcome a severe Zn deficiency and should persist for three to ten years (depending on soil type). Short intervals between repeat applications of Zn will be necessary on heavy and calcareous soils in the high rainfall areas, while seven to ten year intervals will be acceptable in the low rainfall areas. Following an initial soil application of 2 kg Zn/ha repeat applications of 1 kg/ha will probably be sufficient to avoid the reappearance of Zn deficiency in crops and pastures. [56]

A foliar spray per ha of 1.0 kg zinc sulfate heptahydrate + 1.0 kg urea + 1200 mL of non-ionic wetter (1000 g/L) in at least 100 L of water will correct a mild deficiency. One or two sprays will need to be applied within 6–8 weeks of emergence.

Hard water (high in carbonate) will produce an insoluble sediment (zinc carbonate) when the zinc sulfate is dissolved, with the spray mix turning cloudy. Buffer back with L1-700 or Agri Buffa if only hard water is available; zinc oxide products are highly alkaline, with a pH of 9.5–10.5. [57]

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55 GRDC Update Papers. 2016. Detecting and managing trace element deficiencies in crops. https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/02/Detecting-and-managing-trace-element-deficiencies-in-crops

56 GRDC Update Papers. 2016. Detecting and managing trace element deficiencies in crops. https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/02/Detecting-and-managing-trace-element-deficiencies-in-crops

57 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited

IN FOCUS

Chickpea response to zinc, boron and molybdenum application under Mediterranean field conditions

In some growing regions, Chickpea is cultivated on non-irrigated soils with low native fertility. This study was carried out from 2006 to 2008 in Spain, under acid soil field conditions, with the aim of determining whether the application of zinc (Zn), boron (B) and molybdenum (Mo) improved chickpea growth and yield on acid soils. A split-split-plot design with three replications was used. Chickpea responded only to the Zn and Mo applications. At maturity, plants fertilised with Zn and with Mo had a greater total dry matter production and seed yield, mainly due to an increment in pod dry matter. For Zn, the highest yield was obtained with 2 mg Zn per plant (6.80 g plant−1), whereas for Mo the highest yield was obtained with 1 mg Mo per plant (6.73 g plant−1). Interaction was observed between B and Mo, interpreted as indicating that Mo can counteract the effect of B application. [58]

5.11.2 Boron

Key points

- Boron is essential for plant growth, but only needed in very small amounts.
- Soils deficient in boron are often deep sands in high rainfall zones.
- Toxic levels of boron tend to be found in the heavier soils of the Mallee regions.
- Boron toxicity is best managed through the use of crops that exhibit tolerance to the nutrient.

Boron (B) is essential for crop growth and development but in very small quantities. While the precise role of boron in plants is not fully known there is evidence to show that boron is important for cell division, the production of nucleic acids (DNA, RNA), the movement of sugars across membranes and the development of reproductive structures (i.e. pollen tubes, fruit, grain). [59]

For most crops, 1–4 mg-B/kg soil is sufficient to prevent nutrient deficiencies. Less than 0.5 mg-B/kg is rated as marginal to deficient. Boron is generally present in soils as $B_4O_7^{2-}$, $H_2BO_3^-$, HBO_3^{2-}, and BO_3^{3-}. Each of these ionic forms are readily leached under high rainfall conditions. Acid deep sands in higher rainfall regions (>600 mm) where there is little clay and organic matter within the root zone are at most risk of having low boron levels. Symptoms of boron deficiency vary between plants ranging from hollow cavities in vegetable crops, distorted growing tips, discoloration and a 'corky' appearance in fruit and flower and pod abortion in canola. Symptoms are most noticeable in actively growing sites.

Boron toxicity

Soil pH affects the availability of most nutrients. Occasionally, some nutrients are made so available that they inhibit plant growth. For example on some acid soils, Al and Mn levels may restrict plant growth, usually by restricting the rhizobia and so the plant's ability to nodulate.

58 Valenciano, J. B., Boto, J. A., & Marcelo, V. (2011). Chickpea (*Cicer arietinum* L.) response to zinc, boron and molybdenum application under field conditions. New Zealand Journal of Crop and Horticultural Science, 39(4), 217-229.

59 Soil Quality.org. Boron – Western Australia. http://www.soilquality.org.au/factsheets/boron

Boron toxicity in Australia is mainly confined to the low rainfall (<550 mm/yr) Mallee vegetation communities of, South Australia, Victoria, and Western Australia. The soils typically contain highly alkaline (pH >8) and sodic clay subsoils which are poorly leached and have boron concentrations >12 mg-B/kg of soil. Often boron toxic soils have formed from marine sediments or boron rich minerals including tourmaline.

Chickpeas are considered sensitive to boron toxicity and occurs on many of the alkaline soils of the southern cropping areas. Symptoms show as a yellowing or dying of the tips and margins of the leaves, with the older leaves being more severely affected than younger leaves (Photo 11). There appears to be little difference in reaction between current varieties. [60]

Photo 11: *Symptoms of boron toxicity in chickpea leaves.*

Source: CSIRO.

Managing boron toxicity can be achieved through leaching, the application of amendments and using tolerant varieties. Irrigating to encourage leaching is highly effective. However, in the absence of irrigation water, amending soils with gypsum can increase water infiltration in sodic clays and consequently leach boron deeper into the soil. Dryland trials have shown high rates of gypsum over 20 years can leach boron approximately 10–20 cm. In some circumstances, foliar sprays of zinc have been shown to alleviate boron toxicity, although the interaction between boron and zinc is poorly defined.

Boron testing

Soil testing is considered the best method for determining the presence of boron deficiency or toxicity. However due to the high spatial variability in soil boron, testing needs to be done strategically in areas of high and low plant production and throughout the root zone. Due to the mobile characteristics of boron in soil, the most accurate determination of boron status is to sample soil to depth. Hot water extraction in 0.01 M $CaCl_2$ solution is the recommended method for determining soil boron. Plant tissue testing is less reliable as critical limits cannot be easily determined due to the uneven accumulation of boron in plant tissues, variation in boron uptake at different growth stages and the leaching of boron from plant tissue during rainfall. Seed testing is seen as a more reliable method for determining potential boron

60 Pulses Australia. Chickpea Production: Southern and Western Region. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

toxicity. Grain with more than 3 mg-B/kg is likely to have been grown in boron toxic soils. [61]

5.11.3 Iron

Chickpeas vary in their sensitivity to iron (Fe) deficiency. Considerable yield losses due to iron deficiency chlorosis may occur when susceptible varieties are grown in calcareous soils with high pH. Iron deficiency generally results in stunted growth, with deficient plants showing poor nodulation. [62]

Major problems with Fe deficiency have largely been overcome through plant breeding. Iron deficiency symptoms tend to be transient, with the crop making a rapid recovery once the soil begins to dry out following a waterlogging event.

Iron deficiency is observed occasionally on alkaline, high-pH soils. It is usually associated with a waterlogging event following irrigation or heavy rainfall, and is attributed to interference with iron absorption and translocation to the foliage.

A mixture of 1 kg/ha of iron sulfate + 2.5 kg/ha of crystalline sulfate of ammonia (not prilled) + 200 mL of non-ionic wetter added to 100 L water has been successfully used to correct Fe deficiency.

The addition of sulfate of ammonia will improve absorption of Fe, with a significantly better overall response. [63]

Symptoms

- Plants display deficiency symptoms first on younger leaves which turn bright yellow then white, while older leaves remain dark green (Photo 12).
- As symptoms advance, white necrotic areas develop in the distal half of the leaflets in young leaves.
- In the later stage of deficiency, the white necrotic areas enlarge and the leaves wither, die and drop off. [64]

Photo 12: *Leaflets of younger leaves are uniformly bright yellow to white, while older leaves remain dark and healthy.*

Source: Kumar, P., & Sharma, M. K. (Eds.). (2013). Photo: Dr P. Kumar.

5.12 Nutritional deficiencies

Many soils in the cropping zone of southern Australia are deficient in trace elements in their native condition. Despite many decades of research into trace element management, crops can still be found to be deficient in one or more of these trace elements. Just because trace element deficiencies have not been prevalent in recent years, does not mean they will not return.

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61 Soil Quality.org. Boron – Western Australia. http://www.soilquality.org.au/factsheets/boron

62 Kumar, P., & Sharma, M. K. (Eds.). (2013). Nutrient Deficiencies of Field Crops: Guide to Diagnosis and Management. CABI.

63 Pulse Australia (2013) Northern chickpea best management practices training course manual http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/northern-guide Limited.

64 Kumar, P., & Sharma, M. K. (Eds.). (2013). Nutrient Deficiencies of Field Crops: Guide to Diagnosis and Management. CABI.

There is increasing concern in some districts that trace element deficiencies may be the next nutritional barrier to improving productivity. This is because current cropping systems are exporting more nutrients to the grain terminal than ever before.

Making use of the crop nutrition information available to you

As part of the Grains Research and Development Corporation (GRDC) More Profit from Crop Nutrition (MPCN) extension and training for the southern region project (BWD00021), BCG, in conjunction with other grower groups has been hosting nutrition events across the southern region since 2012.

Many key nutrition areas are being investigated through the MPCN initiative; however, there are a few immediate resources available to advisers to help with understanding nutrition and giving such advice.

Useful resources:

- eXtension Aus—Crop Nutrition: Connecting the lab and the paddock in crop nutrition. Providing updates on the latest research, and articles focusing on strategic management of crop nutrition in the current season. @AuCropNutrition
- BFDC—Better Fertiliser Decisions for Cropping: Fertiliser decisions made by grain growers should all start with, and rely on, knowledge of the fertility status of paddocks. These decisions need to account for the nutrient requirements of plants for growth, nutrient availability in soils, and nutrient losses that can occur during crop growth (e.g. de-nitrification or erosion).
- The Making Better Fertiliser Decisions for Cropping Systems in Australia (BFDC) provides the fertiliser industry, agency staff and agribusiness advisors with knowledge and resources to improve nutrient recommendations for optimising crop production. BFDC is recognised by the Fertiliser Industry Federation of Australia as the best available data for supporting the decision tools that fertiliser industry members use to formulate recommendations.
- MPCN—Extension and training for the Southern region. [65]

65 GRDC Update Papers. 2016. Making use of the crop nutrition information available to you. https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/02/Making-use-of-the-crop-nutrition-information-available-to-you

FAQ

Weed control

Key messages

- Chickpeas are poor competitors with weeds because of slow germination and early growth.
- Weed control is essential if the chickpea crop is to make full use of in-crop rainfall and stored soil moisture and nutrients and to prevent weed seeds from contaminating the grain sample at harvest.
- Weed management should be planned well before planting, with chemical and non-chemical control options considered.
- There are limited options for pre-emergent and post-emergent weed control.
- Broadleaf weeds must be heavily targeted in the preceding crop and/or fallow. Always assess the broadleaf weed risk prior to planting.
- Chickpeas should always be planted into planned paddocks that have low weed populations.
- Chickpeas are late-maturing compared with other pulses; hence, crop-topping to prevent ryegrass and other weed seed-set is not possible.

Weeds are estimated to cost Australian agriculture AU$2.5-4.5 billion per annum, with winter cropping systems alone, baring a $1.3 billion cost (Photo 1). Consequently, any practice that can reduce the weed burden is likely to generate substantial economic benefits to growers and the grains industry (Table 1).

The latest recommendations from research in the high rainfall zones of southern Australia can be summed up as: sow a competitive crop early, on the first opening rain, with pre-emergent herbicide; sow the cleanest paddocks last; and implement harvest weed seed control. [1]

Photo 1: *Glyphosate resistant population of sowthistle show dead susceptible plants in the foreground.*

Photo: Graham Charles, Source: GRDC

1 WeedSmart. Gearing up to use pre-emergent herbicides. http://www.weedsmart.org.au/bulletin-board/gearing-up-to-use-pre-emergent-herbicides/

MORE INFORMATION

Impact of weeds on Australian grain production

GRDC Weed management hub.

Table 1: *Yield loss and revenue loss for residual weeds in all crops.*

Area	Residual weeds for all crops			
	Yield loss (t)	Revenue loss	Yield loss (t/ha)	Revenue loss (per hectare)
SA Mid-north – Lower Yorke Eyre	140,936	$37.5m	0.08	$21.67
SA Victorian Bordertown – Wimmera	95,724	$24.6m	0.05	$13.27
SA Victorian Mallee	72,895	$17.8m	0.02	$5.90
Victorian high-rainfall and Tasmanian grain	23,443	%5.9m	0.07	$18.60

Source: GRDC.

Early research indicates that chickpea seed yield can be reduced by 81% and straw yields by 63% when fields remained weed infested until harvest, compared with weed-free conditions throughout the growing season. The critical period of weed interference has been suggested to be between 35 and 49 days after emergence in chickpea. [2]

Weed control is essential if the chickpea crop is to make full use of stored summer rainfall, and in order to prevent weed seeds from contaminating the grain sample at harvest. Weed management should be planned well before planting, with chemical and non-chemical control options considered.

Chickpea crops are poor competitors with weeds because of their slow emergence and growth during winter. Kabuli chickpea competes poorly with weeds, particularly broad-leaved weeds such as radish, mustard, capeweed, and doublegee. Effective weed control is essential to prevent yield loss and to avoid the build-up of troublesome weeds in the rotation. Because of the slow growth and open canopy in chickpeas, narrow or wide row spacing (30 v. 70 cm) makes little difference to the chickpea plant's ability to compete with weeds. The weed-control strategy for growing a successful chickpea crop is based on substantially reducing the viable weed seedbank in the soil before the crop emerges, as post-emergence weed control options are limited. Broadleaf weed control options can be very limited in chickpeas, and this is a reason producers commonly give for not growing chickpeas.

The over-use of particular groups of herbicides through the rotation can lead to herbicide resistance, which has occurred in grass weeds and now some broadleaf weeds. To avoid herbicide resistance, weed management through the rotation should aim to minimise the need for herbicides, to avoid the overuse of any one group of herbicides and to use the least selective herbicide. Effective grass control in the chickpea crop has the benefit of reducing the need for selective grass herbicides in the following cereal year. [3]

Weed control is important because weeds can reduce yield and:

- rob the soil of valuable stored moisture
- rob the soil of nutrients
- cause issues at sowing time, restricting access for planting rigs (especially vine-type weeds such as melons, tar vine or bindweed, which wrap around tines)
- cause problems at harvest
- increase moisture levels of the grain sample (green weeds)
- contaminate the sample

2 Al-Thahabi, S. A., Yasin, J. Z., Abu-Irmaileh, B. E., Haddad, N. I., & Saxena, M. C. (1994). Effect of weed removal on productivity of chickpea (*Cicer arietinum* L.) and lentil (Lens culinaris Med.) in a Mediterranean environment. Journal of Agronomy and Crop Science, 172(5), 333-341.

3 Pulses Australia. Chickpea Production: Southern and Western regions. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

VIDEOS

1. Grains research updates 2015: Problem weeds in Chickpea and Fallow.

- prevent some crops being grown where in-crop herbicide options are limited, i.e. broadleaf crops
- be toxic to stock
- carry disease
- host insects

Recent climate change impacts and implications

The rainfall patterns across south-eastern Australia over the past decade have been dominated by a strong and highly significant autumn rainfall decline. At the same time, the trend towards earlier seeding to improve Water Use Efficiency has meant that lower levels of grasses are germinating before sowing increasing the selection pressure on soil residual herbicides and post sowing herbicides. On Eyre Peninsula, there has been a selection for later germinating barley grass. [4]

6.1.1 Critical period for weed control

One study has found that chickpea must be kept weed-free between the five-leaf and full flowering stages (24–48 DAE) and from the four-leaf stage to beginning of flowering (17–49 DAE) in order to prevent >10% seed yield loss. The overall conclusion of this research was that chickpea should be weed free from 17–60 DAE and outside of this time-frame weeds are unlikely to significantly impact on yield. [5]

6.1 Integrated weed management (IWM)

There are very effective strategic and tactical options available to manage weed competition that will increase crop yields and profitability. Weeds with herbicide resistance are an increasing problem in grain cropping enterprises. The industry and researchers advise that growers adopt integrated weed management (IWM) to reduce the damage caused by herbicide-resistant weeds.

The following five-point plan will assist in developing a management plan in each and every paddock.

1. Review past actions and history.
2. Assess current weed status.
3. Identify weed management opportunities.
4. Match opportunities and weeds with suitably effective management tactics.
5. Combine ideas into a management plan. Use of a rotational plan can assist.

Integrated weed management (IWM) is a system for long-term weed management and is particularly useful for managing and minimising herbicide resistance.

An integrated weed management plan should be developed for each paddock or management zone.

In an IWM plan, each target weed is attacked using tactics from several tactic groups (see links below). Each tactic provides a key opportunity for weed control and is dependent on the management objectives and the target weed's stage of growth. Integrating tactic groups reduces weed numbers, stops replenishment of the seedbank, and minimises the risk of developing herbicide-resistant weeds.

The Grains Research and Development Corporation (GRDC) supports integrated weed management. Download the Integrated Weed Management Manual.

IWM tactics

- Reduce weed seed numbers in the soil
- Controlling small weeds
- Stop weed seed set

4 PIR.SA. Integrated weed management. Cropping weed management.

5 Mohammadi, G., Javanshir, A., Khooie, F. R., Mohammadi, S. A., & Zehtab Salmasi, S. (2005). Critical period of weed interference in chickpea. Weed research, 45(1), 57-63.

- Reduce weed seed numbers in the soil
- Hygiene - prevent weed seed introduction
- Agronomic practices and crop competition

Successful weed management also relies on the implementation of the best agronomic practices to optimise crop growth. Basic agronomy and fine-tuning of the crop system are the important steps towards weed management.

There are several agronomic practices that improve crop environment and growth, along with the crop's ability to reduce weed competition. These include crop choice and sequence, improving crop competition, planting herbicide-tolerant crops, improving pasture competition, using fallow phases, and controlled traffic or tramlining. [6]

Because management of herbicide resistance is case specific, it is difficult to prescribe 'recipes' for how to manage a problem. Instead, you need to understand your situation and choose from a range of methods, such as those outlined below.

Choose a method from the IWM tool box. Consider how these might fit into your farming system and seek advice from your local agronomist. These methods allow you to keep weed populations under control and delay the onset of resistance.

Method 1. Autumn tickle: use light scarification to stimulate weed germination, then spray (paying attention to rotating your chemical groups) before sowing.

Method 2. Barley for weed control: barley is competitive against weeds and use of pre-emergence herbicides is effective. Its shorter growing season also allows for pre-sowing weed control.

Method 3. Catching: use a bin attachment on your harvester to collect weed seed. Then burn the seed. Harvest weed seed management strategies are increasingly being implemented.

Method 4. Crop-topping: Use a non-selective herbicide (paraquat-based) to mature or near-mature crops to reduce weed seed set. However, crop-topping is difficult to implement in chickpeas and is not common practice. Crop-topping in pulse crops can be very effective—weed wiping weeds in lentil crops as a prelude to crop-topping is also effective.

Method 5. Cultivation: Use cultivation to kill germinated weeds.

Method 6. Delayed sowing: Delay sowing for two or more weeks so that additional weeds can be killed by non-selective herbicide. However, yield penalties need to be considered before altering sowing dates.

Method 7. Double-knock strategy: Use a glyphosate application followed by paraquat-based application to control weeds before sowing.

Method 8. Harvest low, no spread, burn: This method has three stages: harvest the crop lower than usual, put the residue (containing weeds) into narrow rows for burning (allows for hotter fire).

Method 9. Hay: Use crop for hay. Trials in southern Australia showed that hay making was most effective—reducing seeds per square metre. Hay cutting alone doesn't guarantee success; you also need to graze or spray-top after it re-shoots to prevent seed on regrowth, and take care when feeding hay out that ryegrass seeds aren't spread to other paddocks.

Method 10. Heavy grazing: Weed seed set is reduced by timely intense grazing of paddocks not sown to crop (and seedbank is reduced).

Method 11. High crop sowing rate: Used to produce a higher crop plant density to reduce yield loss due to weeds and to suppress weed seed production.

6 DAFWA. (2016). Crop weeds: Integrated Weed Management (IWM). https://www.agric.wa.gov.au/grains-research-development/crop-weeds-integrated-weed-management-iwm

MORE INFORMATION

Herbicide resistance and integrated weed management (iwm) in crops and pasture monitoring tools

GRDC IWM Hub.

Weed Seed Wizard

Method 12. Manuring: Use the crop for 'green manure' before it matures to prevent weed seed set and increase organic matter.

Method 13. Mechanical pasture top: Slash the pasture before weed maturity.

Method 14. Spray-topping: Use a low-rate of non-selective herbicide applied to pastures to reduce weed seed set.

Method 15. Careful consideration of rotations (pasture phase instead of continuous cropping). A two-year (or more) pasture phase treated to reduce weeds before it goes back into crop. A pasture phase longer than two years is very effective (one year is not enough to reduce seedbank). It should be noted that it is not the pasture phase itself that helps to manage weeds, but it is what the phase allows you to do in addition, e.g. grazing and pasture topping. Crop rotations need to be managed carefully. Continuous cropping requires strong planning and management practices.

Method 16. Stubble burning: Stubble is burned in autumn to reduce viable weed seeds (but reduces organic matter).

Method 17. Windrowing for weed control: cutting crop near to full maturity and leave to dry in rows to reduce seed shatter (usually done in canola). Can be done in other crops but earlier than usual and lower than normal. [7]

Weed Seed Wizard

The Weed Seed Wizard helps growers to understand and manage weed seedbanks on farms across Australia's grain-growing regions.

Weed Seed Wizard is a computer simulation tool that uses paddock-management information to predict weed emergence and crop losses. Different weed-management scenarios can be compared to show how different crop rotations, weed control techniques, and irrigation, grazing and harvest management tactics can affect weed numbers, the weed seedbank and crop yields.

The Wizard uses farm-specific information, and users enter their own farm-management records, their paddock soil type, local weather and one or more weed species. The Wizard has numerous weed species to choose from, including annual ryegrass, barley grass, wild radish, wild oat, brome grass and silver grass in the southern states.

A free download is available from: https://www.agric.wa.gov.au/weed-seed-wizard-0.

The Weed Seed Wizard is helping farm advisers and their grain-grower clients make decisions that will reduce weed seedbanks and the cost of controlling those weeds.

6.2 Planting control strategies

Pulses grown in rotation with cereal crops offer farmers opportunities to easily control grassy weeds with selective herbicides that cannot be used in the cereal years. An effective kill of grassy weeds in the pulse crop will reduce root disease carry over and provide a "break crop" benefit in the following cereal crop. Grass control herbicides are now available which will control most grassy weeds in pulses. Volunteer cereals can also be controlled with some of these herbicides. Simazine alone and in mixtures with trifluralin in pulse crops can be used to control some other grasses (such as silver grass) that are not readily controlled by the specific grass herbicides. [8]

Do not sow chickpea into a pasture paddock where broadleaf weed pressure will be high. For best results, sow chickpea into paddocks with low broadleaf weed populations. Make the most of opportunities to reduce broadleaf weeds in the preceding crop when weed control is likely to be more effective, cheaper, and cause

7 Agriculture Victoria. HERBICIDE RESISTANCE AND INTEGRATED WEED MANAGEMENT (IWM) IN CROPS AND PASTURE MONITORING TOOLS. http://agriculture.vic.gov.au/agriculture/farm-management/business-management/ems-in-victorian-agriculture/environmental-monitoring-tools/herbicide-resistance

8 GRDC Grain Legume Handbook – Weed control.

less damage to that crop. Delaying chickpea sowing until after a germination of broadleaf weeds also assists in areas or seasons where this is possible. [9]

The use of rotations that include both broadleaf and cereal crops may allow an increased range of chemicals—say three to five MOAs—or non-chemical tactics such as cultivation or grazing.

Strategic cultivation can provide control of herbicide-resistant weeds and those that continue to shed seed throughout the year. It can be used to target large, mature weeds in a fallow, for inter-row cultivation in a crop, or to manage isolated weed patches in a paddock. Take into consideration the size of the existing seedbank and the increased persistence of buried weed seed, but never rule it out. [10]

It is important that broadleaf populations are considered when selecting a paddock for chickpea production. Broadleaf weeds should be heavily targeted in the preceding wheat or barley crop or fallow, bearing in mind the herbicide residual effects. Paddocks with severe broadleaf weed infestation should be avoided. [11] If broadleaf weeds that are not well-controlled by registered broadleaf herbicides are present, then consider altering the cropping rotation until the weed species is controlled.

6.2.1 Managing wild oats in chickpeas

Wild oats (*Avena fatua* and *A. ludoviciana*) represent a large cost to cropping (Photo 2). Wild oats are highly competitive and when left uncontrolled can reduce wheat yields by up to 80%. Greatest yield loss occurs when the wild oat plants emerge at the same time as the crop. Chickpea rotations provide an opportunity to control wild oats, which is otherwise a costly weed in a wheat-based system. However, care should be taken to ensure that surviving weeds are identified and removed to reduce the chance of resistance developing. Herbicide-resistant wild oats are becoming a key threat to sustainable farming systems. Herbicide resistance in wild oats poses management problems in any crop where these herbicides have previously been relied upon, but the threat appears greater to chickpea production. Chickpeas are most at risk because they are a poorly competitive crop and often produced on wide rows. In addition, they have only Group A herbicides available for post-emergent control. Effective use of crop rotation must be made to assist in management of wild oats. This will allow the use of the winter fallow and other effective herbicides (differing MOAs including knockdowns) as well as improved crop competition to reduce seed-set of wild oats. [12] Factor® (butroxydim) was not effective as a knockdown for wild oats in chickpeas. Mixtures of Verdict® (haloxyfop) plus Status® (clethodim) or 500 mL/ha of Status® proved the most effective mixes for knockdown control of wild oats. [13] None of the newer herbicides are registered for wild oat control.

9 Pulses Australia. Chickpea Production: Southern and Western regions. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

10 GRDC (2012) Herbicide resistance. Cropping with herbicide resistance. GRDC Hot Topics, http://www.grdc.com.au/Media-Centre/Hot-Topics/Herbicide-Resistance

11 DAFF (2012) Chickpea—weed management. Department of Agriculture, Fisheries and Forestry, Queensland, http://www.daff.qld.gov.au/plants/field-crops-and-pastures/broadacre-field-crops/chickpeas/weed-management

12 Storrie (2007) Managing wild oats in chickpeas—our practices must change. Northern Grower Alliance, http://www.nga.org.au/results-and-publications/download/45/australian-grain-articles/weeds-1/wild-oats-inchickpeas-tip-of-the-iceberg-september-2007.pdf

13 GRDC Update Papers. 2015. NGA Chickpea herbicide trials 2014. https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2015/02/NGA-Chickpea-herbicide-trials-2014

Photo 2: *Wild oats have a large ligule with no auricles and the leaves tend to be hairy with a slight bluish hue. The emerging leaf is rolled. Wild oats tend to grow in discrete patches at low to moderate densities (up to 100 plants/m^2) and can be confused with brome grass in the seedling stage.*

Source: DAFWA, Photo: Syngenta.

6.2.2 Row spacing

Why do narrow rows yield more?

- reduced weed competition
- early canopy closure
- increased light interception
- reduced evaporation
- reduced competition between crop plants within the row

The simple reason for reduced ryegrass seed set in narrow row spacing is light interception by the crop (Photo 3). In the '90s, when no-till was being adopted, most growers had little choice but to adopt wide row spacing. Stubble retention and no-till go hand-in-hand, so burning for stubble handling became frowned upon for good reason. Harvester capacity was limited, so harvesting low was out of the question, and seeders struggled to handle stubble. Wide rows were the only option.

Photo 3: *Narrow v. wide row canola in 2009. No light reaching the ground in narrow row spacing plots. Ryegrass germinated about when this photo was taken and was not sprayed due to crop safety concerns. Very low ryegrass seed set in the 9 and 18 cm row spacing treatments (top) compared to 27 and 36 cm treatments (bottom).*

Source: UWA.

In 2016 we have harvesters that can harvest low (10–15 cm) and cut and spread the straw evenly. We also have seeders that can handle more stubble than their predecessors. Many growers in regions that regularly achieve wheat yields of 3 to 4 t/ha have successfully adopted 7" (19 cm) row spacing with both tine and disc machines. It's less convenient than wide rows and costs more, but the benefits outweigh the negatives.

WA trials have been conducted exploring the effect of row spacing in weed control for a variety of crops. Tines were removed from the combine as row spacing widened. In a nutshell, narrow row spacing yielded more and had fewer ryegrass (Tables 2 and 3).

Table 2: *Crop yield (kg/ha).*

Year	Crop	Row Spacing			
		9 cm	18 cm	27 cm	36 cm
2003	Wheat	3210	3317	3099	3049
2004	Wheat	1823	1825	1760	1560
2005	Field pea	1995	2024	1761	1710
2006	Wheat	2585	2631	2358	2216
2007	Barley	366	385	394	441
2008	Chemical fallow	*	*	*	*
2009	Canola	929	887	832	766
2010	Wheat	1273	1077	988	1031
2011	Wheat	2140	2058	1969	1975
2012	Chickpea	176	148	119	141
2013	Wheat	2083	2021	2196	2035

Source: UWA.

Table 3: *Annual ryegrass seed (per m^2) at harvest.*

Year	Crop	Row Spacing			
		9 cm	18 cm	27 cm	36 cm
2003	Wheat	324	296	702	382
2004	Wheat	318	312	757	1001
2005	Field pea	375	558	1930	1581
2006	Wheat	14	18	29	27
2007	Barley	25	54	424	789
2008	Chemical fallow	*	*	*	*
2009	Canola	140	319	3056	3468
2010	Wheat	17	24	36	173
2011	Wheat	159	162	334	552
2012	Chickpea	60	50	135	287
2013	Wheat	2	1	51	171

Source: UWA.

MORE INFORMATION

UWA – Narrow Row spacing – more crop, fewer weeds.

VIDEOS

2. AHRI insight #70: Narrow row spacing, more crop, fewer weeds.

IN FOCUS

Chemical and Non-chemical Weed Control in Wide Row Chickpeas

Organic crops are on the increase worldwide and weed control is one of the main problems since chemicals cannot be used. Wide row lupin sowing systems (greater than 50 cm wide rows) are becoming common and this allows growers to control interrow weeds by inter-row cultivation for organic crops or spraying non-selective herbicides using shielded sprayers. In this study, inter-row shielded spraying was found to be the most effective treatment for annual ryegrass control in the 66 cm wide rows, but future herbicide resistance will be a major limitation. With shielded spraying, some form of intra-row weed control will still be necessary to significantly reduce weed seed set. Automatic tractor steering control would also be essential for commercial growers to adopt shielded spraying. In 2006, inter-row cultivation reduced annual ryegrass biomass by 63% and the number of annual ryegrass heads by 43%. To be most effective, it is suggested that inter-row cultivation should be done relatively early while the weeds are small, and when the soil is relatively warm and dry with rain not predicted for a day or two. In 2006 and 2007, inter-row shielded spraying with glyphosate gave the best ryegrass control averaging 94%. Weed seed head trimming or cutting weeds above the crop prior to weed seed maturity may be a useful non-chemical method to reduce the number of weed seeds set if the weed seed is above the crop canopy and the cutting height is well controlled. Indian hedge mustard (Sisymbrium orientale) seed collected in the 2005 chickpea harvest samples was reduced by around 35% with all trimming treatments. In 2006, the late flower trimming reduced the seed number of wild oats and volunteer wheat in chickpeas. Lupin and chickpea grain yield was slightly reduced by trimming in 2005, but with improved height control did not reduce yields in 2006. Given the difficulties in controlling weeds by the growers due to widespread development of herbicide resistance in these weeds, this novel non-chemical way of weed control is a viable promising option to reduce the soil weed seed bank. [14]

6.3 Herbicides explained

6.3.1 Residual and non-residual

Residual herbicides remain active in the soil for an extended period (months) and can act on successive weed germinations. Residual herbicides must be absorbed through the roots or shoots, or both. Examples of residual herbicides include isoxaflutole, imazapyr, chlorsulfuron, atrazine, and simazine.

The persistence of residual herbicides is determined by a range of factors including application rate, soil texture, organic matter levels, soil pH, rainfall and irrigation, temperature, and the herbicide's characteristics. The persistence of herbicides will affect the enterprise's sequence (a rotation of crops, e.g. wheat–barley–chickpeas–canola–wheat).

Non-residual herbicides, such as the non-selective paraquat and glyphosate, have little or no soil activity and they are quickly deactivated in the soil. They are either

14 Riethmuller, G. P., Hashem, A., & Pathan, S. M. (2009). Chemical and non-chemical weed control in wide row lupins and chickpeas in Western Australia. Australian Journal of Multi-disciplinary Engineering, 7(1), 15-26.

broken down or bound to soil particles, becoming less available to growing plants. They also may have little or no ability to be absorbed by roots.

6.3.2 Post-emergent and pre-emergent

These terms refer to the target and timing of herbicide application. Post-emergent refers to foliar application of the herbicide after the target weeds have emerged from the soil, whereas pre-emergent refers to application of the herbicide to the soil before the weeds have emerged. [15]

6.4 Mode of Action (MOA)

Herbicides have been classified into a number of 'groups'. The group refers to the way a chemical works—their different chemical make-up and mode of action (see Herbicide Mode of Action Groups, for a full list of options).

Resistance has developed primarily because of the repeated and often uninterrupted use of herbicides with the same mode of action. Selection of resistant strains can occur in as little as 3–4 years if attention is not paid to resistance management. Remember that the resistance risk remains for products having the same MOA. If you continue to use herbicides with the same MOA and do not follow a resistance-management strategy, problems will arise.

6.4.1 MOA labelling

In order to facilitate management of herbicide-resistant weeds, all herbicides sold in Australia are grouped by MOA. The MOA is indicated by a letter code on the product label. The MOA labelling is based on the resistance risk of each group of herbicides. Australia was the first country to introduce compulsory MOA labelling on products, and the letters and codes used in Australia are unique. Labelling is compulsory and the letters and codes reflect the relative risk of resistance evolving in each group. Since the introduction of MOA labelling in Australia, other countries have adopted MOA classification systems; however, caution is advised if cross-referencing MOAs between Australia and other countries, as different classification systems are used. The herbicide MOA grouping and labelling system in Australia was revised in 2007. This is the first major revision of the classification system since its introduction.

The original groupings were made based on limited knowledge about MOAs. Groupings have been changed to improve the accuracy and completeness of the MOAs to enable more informed decisions about herbicide rotation and resistance management. The general intent of groups based on their risk has not changed.

6.4.2 Grouping by mode of action and ranking by resistance risk

Growers and agronomists are now better assisted to understand the huge array of herbicide products in the marketplace in terms of MOA grouping and resistance risk by reference to the MOA chart. All herbicide labels now carry the MOA group clearly displayed, such as:

Group	**G**	**Herbicide**

Know your herbicide groups to make use of this labelling.

Not all MOA groups carry the same risk for resistance development; therefore, specific guidelines for Groups E, G, H, N, O, P, and R have not been developed because there are no recorded cases of weeds resistant to members of these groups in Australia.

15 T McGillion, A Storrie (Eds) (2006) Integrated weed management in Australian cropping systems—A training resource for farm advisors. Section 4: Tactics for managing weed populations. CRC for Australian Weed Management, Adelaide, https://grdc.com.au/__data/assets/pdf_file/0030/48639/iwmmc-pdf.pdf.pdf

Products represented in Group A (mostly targeted at annual ryegrass and wild oats) and Group B (broadleaf and grass weeds) are HIGH RESISTANCE RISK herbicides, and specific guidelines are written for use of these products in winter cropping systems.

Specific guidelines are also available for the MODERATE RESISTANCE RISK herbicides: Group C (annual ryegrass, wild radish and silver grass), Group D (annual ryegrass and fumitory), Group F (wild radish), Group I (wild radish and Indian hedge mustard), Group J (serrated tussock and giant Parramatta grass), Group L (annual ryegrass, barley grass, silver grass and cape weed), Group M (annual ryegrass, barnyard grass, fleabane, liverseed grass and windmill grass), Group Q (annual ryegrass), and Group Z (wild oats and winter grass).

Specific guidelines for Group K have been developed due to the reliance on this MOA to manage annual ryegrass, and the possibility of future resistance development. [16]

6.4.3 Specific guidelines for Group A herbicides

The following charts have been compiled from chemical labels on the APVMA web site and PIRSA Spraying charts and in consultation with chemical companies. (Source: CropLife Australia 'Herbicide Resistance Management Strategies—September 2011'.)

Group	A	Herbicide

High resistance risk.

Group A resistance exists in Australia in the grass weeds, including annual ryegrass, wild oats, phalaris, brome grass, crab grass, goose grass and barley grass. Resistance has developed in broadacre and vegetable situations.

Research has shown that as few as six applications to the same population of annual ryegrass can result in the selection of resistant individuals. A population can go from a small area of resistant individuals to a whole-paddock failure in one season.

Fops, dims, and dens are Group A herbicides and carry the same high resistance risk. Where a Group A herbicide has been used on a particular paddock for control of any grass weed, avoid using a Group A herbicide to control the same grass weed in the following season, irrespective of the performance it gave.

Frequent application of Group A herbicides to dense weed populations is the worst scenario for rapid selection of resistance.

Where resistance to a member of Group A is suspected or known to exist, there is a strong possibility of cross-resistance to other Group A herbicides. Therefore, use other control methods and herbicides of other MOA groups in a future integrated approach.

The above recommendations should be incorporated into an integrated weed management (IWM) program. In all cases, try to ensure that surviving weeds from any treatment do not set and shed viable seed. Keep to integrated strategies, including rotation of MOA groups.

16 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

Table 4: *Active ingredients of Group A MOAs.*

Chemical family	Active constituent (first registered trade name)
Group A. Inhibitors of acetyl coA carboxylase (inhibitors of fat synthesis/ACCase inhibitors)	
Aryloxyphenoxypropionates (fops)	Clodinaafop (Topik®), cyhalofop (Barnstorm®), diclofop (Cheetah® Gold, Decision®*, Hoegrass®, Tristar® Advance*), fenoxaprop (Cheetah® Gold*, Tristar® Advance*, Wildcat®), fluazifop (Fusilade®, Fusion®*), haloxyfop (Motsa®*, Verdict®), propaquizafop (Shogun®), quizalofop (Targa®)
Cyclohexanediones (dims)	Butroxydim (Falcon®, Fusion®*), clethodim (Motsa®*, Select®), profoxydim (Aura®), sethoxydim (Cheetah® Gold*, Decision®*, Sertin®), tepraloxydim (Aramo®), tralkoxydim (Achieve®)
Phenylpyrazoles (dens)	Pinoxaden (Axial®)

*This product contains more than one active constituent.

6.4.4 Specific guidelines for Group B herbicides

(Source: CropLife Australia, 'Herbicide Resistance Management Strategies—September 2011'.)

Group	**B**	**Herbicide**

High resistance risk.

Group B resistance exists in Australia in the grass weeds, annual ryegrass, barley grass, brome grass, wild oats, and crab grass and in at least 16 broadleaf weeds including: wild radish, common sowthistle, climbing buckwheat, turnip weed, wild mustard, Indian hedge mustard, prickly lettuce, wild turnip, and African turnip weed. Resistance has developed in broadacre, rice, and pasture situations. With respect to rice, three broadleaf weeds have been discovered: dirty dora, arrowhead, and starfruit.

Research has shown that as few as four applications to the same population of annual ryegrass can result in the selection of resistant individuals and as few as six applications for wild radish. A population can go from a small area of resistant individuals to a whole paddock failure in one season.

Avoid applying more than two Group B herbicides in any four-year period on the same paddock.

Broadleaf weed control

If a pre-emergent application is made with a Group B herbicide for broadleaf weed control, monitor results and, if required, apply a follow-up spray with a non-Group B herbicide for control of escapes and to reduce seed-set.

If a post-emergent application is made with a Group B herbicide for broadleaf weed control, it should preferably be as an APVMA-approved tank-mix with another MOA that controls or has significant activity against the target weed. If no APVMA-approved tank-mix is available, then monitor results and if required, apply a follow-up spray with a non-Group B herbicide for control of escapes and to reduce seed-set.

A Group B herbicide may be used alone on flowering wild radish only if a Group B herbicide has not been previously used on that crop.

Grass-weed control

If there are significant escapes following the herbicide application, consider using another herbicide with a different mode of action or another control method to stop seed-set.

Table 5: *Active ingredients for Group B MOAs.*

Chemical family	Active constituent (first registered trade name)
Group B. Inhibitors of acetolactate synthase (ASL inhibitors)	
Sulfonylureas (SUs)	Azimsulfuron (Gulliver®), bensulfuron (Londax®), chlorsulfuron (Glean®), ethoxysulfuron (Hero®), foramsulfuron (Tribute®), halosulfuron (Sempra®), iodosulfuron (Hussar®), mesosulfuron (Atlantis®), metsulfuron (Ally®, Harmony®*M, Trounce®*, Ultimate Brushweed®* Herbicide), prosulfuron (Casper®*) rimsulfuron (Titus®), sulfometuron (Oust®), sulfosulfuron (Monza®), thifensulfuron (Harmony®*M), triasulfuron (Logran®, Logran® B-Power*) tribenuron (Express®), trifloxysulfuron (Envoke®, Krismat®*)
Imidazolinones (imis)	Imazamox (Raptor®, Intervix®*), imazapic (Flame®, Midas®*, OnDuty®*), imazapyr (Arsenal Xpress®*, Midas®*, OnDuty®*, Intervix®*, Lightning®*), imazethapyr (Spinnaker®, Lightning®*)
Triazolopyrimidines (sulfonamides):	Flumetsulam (Broadstrike®), florasulam (Conclude®*, Torpedo®*, X-Pand®*), metosulam (Eclipse®), pyroxsulam (Crusader®)
Pyrimidinylthiobenzoates	Bispyribac (Nominee®), pyrithiobac (Staple®)

*This product contains more than one active constituent.

6.4.5 Specific guidelines for Group C herbicides

(Source: CropLife Australia 'Herbicide Resistance Management Strategies—September 2011'.)

Group	C	Herbicide

Moderate resistance risk.

Group C resistance exists in Australia in the weeds annual ryegrass, wild radish, liverseed grass, silver grass, stinging nettles, and barnyard grass. Resistance has developed in broadacre, horticultural, and non-crop situations.

CropLife Australia gives specific guidelines for the use of Group C herbicides in TT canola and in winter legume crops, following increasing reports of resistance development.

Avoid using Group C herbicides in the same paddock in consecutive years. Growing TT canola in a paddock treated with triazine herbicides in the previous season is a high resistance risk and is not recommended.

Watch and record for weed escapes, especially in paddocks with a long history of Group C use.

Consult the 'Integrated Weed Management Strategy for TT Canola' for further details. The resistance status of the 'at-risk' weeds should be determined prior to sowing. Always use the label rate of herbicide, whether a single active ingredient (e.g. bromoxynil) or combination of active ingredients is applied (e.g. bromoxynil/MCPA, pyrasulfotole/bromoxynil). Apply to weeds at the labelled growth stage and ensure that no weeds set and shed viable seed. To prevent seed-set, control survivors with a herbicide of different MOA from Group C, or use another weed-management technique.

Table 6: *Active ingredients in Group C MOAs.*

Chemical family	Active constituent (first registered trade name)
Group C. Inhibitors of photosynthesis at photosystem II (PS II inhibitors)	
Triazines	Ametryn (Amigan®*, Primatol Z®, Gesapax® Combi*, Krismat®), atrazine (Gesaprim®, Gesapax® Combi*, Primextra® Gold*), cynazine (Bladex®), prometryn (Gesagard®, Cotoguard®*, Bandit®*), propazine (Agaprop®), simazine (Gesatop®) terbuthylazine (Terbyne®), terbutryn (Amigan®*, Igran®, Agtryne® MA*)
Triazinones	Hexazinone (Velpar® L, Velpar® K4*), metribuzin (Sencor®)
Uracils	Bromacil (Hyvar®, Krovar®*), terbacil (Sinbar®)
Pyridazinones	Chloridazon (Pyramin®)
Phenylcarbamates	Phenmedipham (Betanal®)
Ureas	Diuron (Karmex®, Krovar®*, Velpar® K4*), fluometuron (Cotoran®, Cotoguard®*, Bandit®*), linuron (Afalon®), methabenzthiazuron (Tribunil®), siduron (Tupersan®), tebuthiuron (Graslan®)
Armides	Propanil (Stam®)
Nitriles	Bromoxynil (Buctril®, Buctril® MA*, Barrel®*, Jaguar®*, Velocity®*, Flight®*), ioxynil (Totril®, Actril DS*)
Benzothiadiazinones:	Bentazone (Basagran®, Basagran® M60*)

*This product contains more than one active constituent.

6.4.6 Specific guidelines for Group D herbicides

(Source: CropLife Australia 'Herbicide Resistance Management Strategies—September 2011'.)

Group	D	Herbicide

Moderate resistance risk.

Resistance to Group D herbicides is known for an increasing number of populations of annual ryegrass and fumitory. Resistance has generally occurred after 10–15 years of use of Group D herbicides.

Where possible, avoid the use of Group D herbicides on dense ryegrass populations. Consider using alternative methods of weed control to reduce weed numbers before applying herbicides.

Rotate with herbicides from other MOA. For annual ryegrass, consider rotating trifluralin with products such as Boxer Gold®.

These recommendations should be incorporated into an IWM program. Try to ensure that surviving weeds from any treatment do not set and shed viable seed. Use integrated strategies, including rotation of MOA groups.

Table 7: *Active ingredients of Group D MOAs.*

Chemical family	Active constituent (first registered trade name)
Group D. Inhibitors of microtubule assembly	
Dinitroanilines (DNAs)	Oryzalin (Surflan®, Rout®*), pendimethalin (Stomp®), prodiamine (Barricade®), trifluralin (Treflan®)
Benzoic acids	Chlorthal (Dacthal®, Prothal®*)
Benzamides	Propyzamide (Kerb®)
Pyridines	Dithiopyr (Dimension®), thiazopyr (Visor®)

*This product contains more than one active constituent.

6.4.7 Specific guidelines for Group F herbicides

(Source: CropLife Australia 'Herbicide Resistance Management Strategies—September 2011'.)

Group	F	Herbicide

Moderate resistance risk.

Resistance to Group F herbicides is known for a small number of populations of wild radish. Resistance has generally occurred after a long history of use of Group F herbicides. The number of populations with Group F resistance is increasing following increased use of these herbicides.

Group F includes herbicides that reduce carotenoid biosynthesis through inhibition of phytoene desaturase (PDS).

Avoid applying Group F herbicides in any two consecutive years unless one application is a mixture with a different MOA that is active on the same weed, or a follow-up spray is conducted (using a different MOA) to control escapes. Always use the label rate of herbicide, whether a single active ingredient (e.g. diflufenican) or combination of active ingredients is applied (e.g. diflufenican/MCPA, picolinafen/MCPA). Apply to weeds at the labelled growth stage and ensure that no weeds set and shed viable seed. To prevent seed-set, control survivors with a herbicide of different MOA from Group F, or use another weed-management technique.

If applicable, apply a follow-up spray with a non-Group F herbicide for control of escapes and to reduce seed-set. Aim to ensure that surviving weeds from any treatment do not set and shed viable seed.

These recommendations should be incorporated into an IWM program. Try to ensure that surviving weeds from any treatment do not set and shed viable seed. Use integrated strategies including rotation of MOA groups.

Table 8: *Active ingredients for Group F MOAs.*

Chemical family	Active constituent (first registered trade name)
Group F. Bleachers: Inhibitors of carotenoid biosynthesis at the phytoenede saturase step (PDS inhibitors)	
Nicotinanilides	Diflufenican (Brodal®, Jaguar®*, Tigrex®*, Chipco Spearhead®*)
Picolinamides	Picolinafen (Paragon®*, Sniper®, Flight®)
Pyridazinones	Norflurazon (Solicam®)

*This product contains more than one active constituent.

6.4.8 Specific guidelines for Group I herbicides

(Source: CropLife Australia 'Herbicide Resistance Management Strategies—September 2011'.)

Group	I	Herbicide

Moderate resistance risk.

Resistance to Group I herbicides is known for a number of populations of wild radish and Indian hedge mustard. Resistance has occurred after a long history of use of Group I herbicides. The number of populations with Group I resistance is increasing.

It is of particular concern that in addition to Group I resistance in wild radish, which is the most important broadleaf weed in broadacre agriculture, some populations are cross-resistant to other MOAs, e.g. Group F herbicides, which can be important for control of wild radish in lupins where other selective, non-Group I options are limited. Because of the long soil life of wild radish seed, measures to reduce the return of seed to the soil would be useful for this weed. Wild radish seed that is confined to the top 5 cm soil has a shorter life than seed buried deeper.

As a rule, in situations of high resistance risk:

- Avoid applying two applications of Group I herbicides alone onto the same population of weeds in the same season.
- Where possible, combine more than one MOA in a single application. Each product should be applied at rates sufficient for control of the target weed alone to reduce the likelihood of weeds resistant to the Group I herbicide surviving.

These recommendations should be incorporated into an IWM program. Try to ensure that surviving weeds from any treatment do not set and shed viable seed. Use integrated strategies, including rotation of MOA groups.

Table 9: *Active ingredients of Group I MOAs.*

Chemical family	Active constituent (first registered trade name)
Group I. Disruptors of plant cell growth (synthetic auxins)	
Phenoxycarboxylic acids (phenoxys)	2,4-D (Amicide®, Actril DS®*, Pyresta®*), 2,4-DB (Trifolamine®), dichlorprop (Lantana 600®), MCPA (MCPA, Buctril® MA*, Banvel M®*, Conclude®*, Midas®*, Paragon®*, Tigrex®*, Barrel®*, Tordon 242®*, Basagran® M60*, Chipco Spearhead®*, Agtryne® MA*, Precept®*, Flight®*) MCPB (Legumine®), mecoprop (Mecopropamine®, Mecoban®, Methar Tri-Kombi®*)
Benzoic acids	Dicamba (Banvel®, Banvel M®*, Barrel®*, Mecoban®, Methar Tri-Kombi®*)
Pyridine carboxylic acids (pyridines)	Aminopyralid (Hotshot®*,Grazon Extra®*), clopyralid (Lontrel®, Torpedo®*, Chipco Spearhead®*), fluroxypyr (Starane®, Hotshot®*), picloram (Tordon®, Tordon 242®*, Grazon®*, Grazon Extra®*), triclopyr (Garlon®, Grazon®*, Grazon Extra®*, Ultimate Brushweed®* Herbicide)

*This product contains more than one active constituent.

6.4.9 Specific guidelines for Group J herbicides

(Source: CropLife Australia 'Herbicide Resistance Management Strategies—September 2011'.)

Group	J	Herbicide

Moderate resistance risk.

There are isolated cases of weeds resistant to Group J in Australia. Two populations of serrated tussock and six populations of giant Parramatta grass are confirmed resistant to flupropanate.

To assist in delaying the onset of resistance, consider alternating with herbicides from other MOA.

The recommendations should be incorporated into an IWM program. Try to ensure surviving weeds from any treatment do not set and shed viable seed. Use integrated strategies, including rotation of MOA groups.

Table 10: *Active ingredients of Group J MOAs.*

Chemical family	Active constituent (first registered trade name)
Group J. Inhibitors of fat synthesis (not ACCase inhibitors)	
Chlorocarbonic acids	2,2-DPA (Dalapon®), flupropanate (Frenock®)
Thiocarbamates	EPTC (Eptam®), molinate (Ordram®), pebulate (Tillam®), prosulfocarb (Boxer® Gold*), thiobencarb (Saturn®), triallate (Avadex®), vernolate (Vernam®)
Phosphorodithioates	Bensuilde (Prefar®)
Benzofurans	Ethofumesate (Tramat®)

*This product contains more than one active constituent.

6.4.10 Specific guidelines for Group K herbicides

(Source: CropLife Australia 'Herbicide Resistance Management Strategies—September 2011'.)

Group	K	Herbicide

Moderate resistance risk.

Resistance to Group K herbicides is possible in Australia and may develop in broadacre situations.

Where possible, avoid the use of Group K herbicides on dense populations of ryegrass. Consider using alternative methods of weed control to reduce weed numbers before applying herbicides.

Rotate with herbicides from other modes of action. The recommendations should be incorporated into an IWM program. Try to ensure surviving weeds from any treatment do not set and shed viable seed. Use integrated strategies, including rotation of MOA groups.

Table 11: *Active ingredients for Group K MOAs.*

Chemical family	Active constituent (first registered trade name)
GROUP K. Inhibitors of cell division/inhibitors of very long chain fatty acids (VLCFA inhibitors).	
Acetamides	Napropamide (Devrinol®)
Chloroacetemides	Dimethenamid (Frontier®-P), metolachlor (Boxer® Gold*, Dual® Gold, Primextra® Gold*), propachlor (Ramrod®, Prothal®*)
Isoxazoline	Pyroxasulfone (Sakura®)

*This product contains more than one active constituent.

6.4.11 Specific guidelines for Group L herbicides

(Source: CropLife Australia 'Herbicide Resistance Management Strategies—September 2011'.)

Group	L	Herbicide

Moderate resistance risk.

Group L resistance exists in Australia in annual ryegrass, barley grass (two species), silver grass, cape weed, and square weed. Most instances have occurred in long-term lucerne stands treated regularly with a Group L herbicide, but Group L-resistant barley grass has also occurred in no-till situations.

The following factors are common to all cases of Group L resistance:

- A Group L herbicide is the major or only herbicide used.

- A Group L herbicide has been used for 12–15 years or more.
- There has been minimal or no soil disturbance following application.

The risk of resistance to Group L herbicides is higher in no-tillage broadacre cropping. Other situations of high resistance risk include irrigated clover pivots, orchards, vineyards, or pure lucerne stands where frequent applications of a Group L herbicide are made each season, cultivation is not used and there is reliance on a Group L herbicide alone for weed control.

Below are strategies to reduce the risk of Group L resistance developing in situations of high resistance risk.

No-tillage

Rotate Group L herbicides with other knockdown herbicides with a different mode of action.

Consider utilising the double-knock technique, with glyphosate sprayed first followed within 1–7 days by a paraquat application. A full label rate for the weed size targeted should be used for the paraquat application for resistance management. Consider occasional mechanical cultivation to aid weed control.

Lucerne

If using a Group L herbicide for winter cleaning, where possible include another MOA, e.g. diuron (Group C).

Use alternative MOA to selectively control grass and broadleaf weeds. Rotate Group L herbicides with other knockdown herbicides with a different MOA prior to sowing lucerne and prior to sowing future crops in that paddock.

Horticulture

Rotate Group L herbicides with other knockdown herbicides with a different MOA. Where possible, use residual herbicides (that are effective on the same weeds as the Group L herbicides) where applicable, either alone or in mixture with Group L herbicides. Where possible, use an alternative MOA to selectively control grass and broadleaf weeds. Consider using the double-knock technique, with glyphosate sprayed first followed within 1–7 days by a paraquat application. A full label rate for the weed size targeted should be used for the paraquat application for resistance management.

These recommendations should be incorporated into an IWM program. Try to ensure surviving weeds from any treatment do not set and shed viable seed. Always try to apply herbicides to the smallest weed density. Use integrated strategies, including rotation of MOA groups.

Table 12: *Active ingredients of Group L MOAs.*

Chemical family	Active constituent (first registered trade name)
Group L. Inhibitors of photosynthesis at photosystem I (PSI inhibitors)	
Bipyridyls	Diquat (Reglone®, Spray.Seed®*), paraquat (Gramoxone®, Spray.Seed®*, Alliance®*)

*This product contains more than one active constituent.

6.4.12 Specific guidelines for Group M herbicides

(Source: CropLife Australia 'Herbicide Resistance Management Strategies—September 2011'.)

Group	M	Herbicide

Moderate resistance risk.

Group M resistance occurs in Australia in annual ryegrass, awnless barnyard grass, fleabane, liverseed grass, and windmill grass.

Herbicide resistance to glyphosate was first discovered in annual ryegrass in Australia in 1996. Since then, several new cases of glyphosate resistance in annual ryegrass, awnless barnyard grass, fleabane, liverseed grass, and windmill grass have been confirmed.

The following factors are common to all cases of Group M resistance:

- A Group M herbicide is the major or only herbicide used.
- A Group M herbicide has been used for 12–15 years or more.
- There has been minimal or no soil disturbance following application.

Given the important role of glyphosate in Australian farming systems, the Australian agricultural industry has developed strategies for sustainable use of glyphosate.

For more information, refer to the Australian Glyphosate Sustainability Working Group website.

All cases of glyphosate resistant weeds confirmed to date share three common factors:

1. Intensive (year-to-year) use of glyphosate
2. Lack of rotation of other herbicide modes of action
3. Little or no tillage or cultivation following the application of glyphosate

Several cases of ryegrass resistance to glyphosate have occurred in horticultural and non-cropping situations (e.g. firebreaks, fence lines, driveways, irrigation ditches), with the balance occurring in no-till, broadacre cropping systems.

Given the demonstrated propensity of annual ryegrass to develop resistance to multiple herbicide classes, IWM principles should be incorporated wherever possible to minimise the risk of selecting for glyphosate-resistant ryegrass. Strategies may include the use of cultivation, the double-knock technique (using a full-cut cultivation OR the full label rate of a paraquat-based product (Group L) following the glyphosate (Group M) knockdown application), strategic herbicide rotation, grazing, and baling.

Try to ensure that surviving weeds from any treatment do not set and shed viable seed. Always try to apply herbicides to the smallest weed density. Use the integrated strategies mentioned, including rotation of MOA groups.

Table 13: *Active ingredients of Group M MOAs.*

Chemical family	Active constituent (first registered trade name)
Group M. Inhibitors of EPSP synthase	
Glycines	Glyphosate (Roundup®, Trounce®*,Illico®*, Arsenal Xpress®*, Broadway®*)

*This product contains more than one active constituent.

6.4.13 Specific guidelines for Group Z herbicides

(Source: CropLife Australia 'Herbicide Resistance Management Strategies—September 2011'.)

Group	Z	Herbicide

Moderate resistance risk.

Group Z resistance exists in Australia in wild oats resistant to flamprop. Many of these flamprop-resistant wild oats also show cross-resistance to Group A herbicides. Resistance to endothal is confirmed in winter grass.

To assist in delaying the onset of resistance, rotate with herbicides from other MOAs. Consider using alternative methods of weed control to reduce weed numbers before applying herbicides. These may include summer crop rotations, delayed sowing to control wild oats with a knockdown herbicide, higher seeding rates and brown manuring to stop seed-set.

The recommendations should be incorporated into an IWM program. Try to ensure that surviving weeds from any treatment do not set and shed viable seed. Use integrated strategies, including rotation of MOA groups.

Table 14: *Active ingredients of Group Z MOAs.*

Chemical family	Active constituent (first registered trade name)
Group Z. Herbicides with unknown and probably diverse sites of action	
Arylaminopropionic acids	Flamprop (Mataven L®)
Dicarboxylic acids	Endothal (Endothal®)
Organoarsenicals	DSMA (Methar®), MSMA (Daconate®)

*This product contains more than one active constituent.

Refer to the APVMA website to obtain a complete list of registered products from the PUBCRIS database.

6.4.14 Herbicide use according to growth stage

Table 15: *Herbicide use according to growth stage in chickpeas.*

Herbicide	Use	Growth stage
Group A		
butroxydim	Do not graze or cut for stockfeed within 14 days of application	Vegetative growth stage, prior to flowering, podding and seven weeks prior to harvest
butroxydim + fluazifop	Up to seven weeks before harvest	Vegetative growth stage, prior to flowering, podding and seven weeks prior to harvest
clethodim	Up to full flowering	Vegetative growth stage, prior to flowering, podding
fluazifop	Up to seven weeks before harvest	Vegetative growth stage, prior to flowering, podding and seven weeks prior to harvest
haloxyfop	Second branch through to flowering	Vegetative growth stage, prior to flowering, podding
propaquizafop	Up to 12 weeks before harvest	Vegetative growth stage
sethoxydim	Up to prior to flowering	Vegetative growth stage, prior to flowering, podding
tepraloxydim	Up to 12 weeks before harvest	Vegetative growth stage
Group B		
flumetsulam	Apply from four to six branches and no later than six weeks after emergence	Vegetative growth stage

Source: DPI NSW.

NOTE: For chickpeas, the window for application for selective grass control herbicides (Group As) is generally dictated by regulatory requirements to avoid residues in produce that exceed levels acceptable to various markets. Check the labels for individual herbicides but chickpea crop safety for most Group As is not influenced by growth stage up to at least flowering.

For up to date chemical Withholding Periods and other label information, see the APVMA search facility.

6.4.15 Getting the best results from herbicides

1. Control weeds as early as possible in the first six weeks after sowing.
2. Make sure that the crop and weeds are at the correct growth stage for the herbicide to be used.
3. Do not spray outside the recommended crop growth stages as damage may result.
4. Do not spray when the crop or weeds are under any form of stress such as drought, water logging, extreme cold, low soil fertility, disease or insect attack, or a previous herbicide.
5. Some herbicides should not be used when weeds are wet with rain or dew or if rain is likely to occur within three or four hours.
6. Do not spray in windy conditions (over 10–15 km/hour) as drift from herbicides can cause damage to non-target crops. Herbicide spray can also drift in very calm conditions, especially with air temperature inversions.
7. Use sufficient water to ensure a thorough, uniform coverage regardless of the method of application.
8. Use good quality water. Hard, alkaline or dirty water can reduce the effectiveness of some herbicides.
9. Maintain clean, well-cared for equipment. A poorly maintained spray unit will cost you money in breakdowns, blocked jets, poor results and perhaps worse, crop damage through misapplication.
10. After products such as Atlantis®, chlorsulfuron, Hussar® metsulfuron, or triasulfuron have been used in equipment, it is essential to clean that equipment thoroughly with chlorine before using other chemicals. After using Affinity®, Broadstrike®, or Eclipse® decontaminate with liquid alkali detergent.
11. Seek advice before spraying recently released pulse varieties. They may differ in their tolerance to herbicides. [17]

6.5 Summer fallow weed control

In a winter cropping system, the return on investment from managing weeds in summer fallow (i.e. the period between crops) is high. Economic benefits flow from both extra amounts of high value water and nitrogen, crop establishment benefits and reduced issues with weed vectored disease and insect pests.

Stopping weed growth in the fallow can lead to yield increases in the following crop via several pathways. These include:

- Increased plant available water
- A wider and more reliable sowing window
- Higher levels of plant available N
- Reduced levels of weed vectored diseases and nematodes
- Reduced levels of rust inoculum via interruption of the green bridge
- Reduced levels of diseases vectored by aphids that build in numbers on summer weeds, and
- Reduced weed physical impacts on crop establishment.

How farming country is managed in the months or years before sowing can be more important in lifting water use efficiency (WUE) than in-crop management. Of

17 GRDC (2008). Grain Legume Handbook – Weed control

MORE INFORMATION

Summer fallow weed management for growers in the Southern region

VIDEOS

3. Over The Fence South: Summer weed control saves moisture for winter crops

particularly high impact are strategies that increase soil capture and storage of fallow rainfall to improve crop reliability and yield.

Practices such as controlled traffic farming and long term no-till seek to change the very nature of soil structure to improve infiltration rates and improve plant access to stored water by removal of compaction zones.

Shorter term management decisions can have an equal or even greater impact on how much plant available water (PAW) is stored at sowing. These include decisions such as crop sequence/rotation that dictate the length of the fallow and amount of stubble cover, how effectively fallow weeds are managed, stubble management and decisions to till/not to till at critical times.

While many factors influence how much plant available water is stored in a fallow period, good weed management consistently has the greatest impact. [18]

6.6 Double knock strategies

Getting the best bang for your buck

The use of glyphosate as the first knock followed within one to seven days with the second knock application of paraquat or paraquat + diquat is increasing in southern Australia. A well-timed and executed double knock is a very useful first step to reducing weed pressure and keeping a lid on glyphosate resistant annual ryegrass. Building the double knock treatment into a whole-of-season weed management plan provides opportunities to get more 'bang for your buck'.

The first knock is to kill all plants still susceptible to glyphosate—applying a lower rate risks higher survival rates, increasing the pressure on the second knock products. The second knock of Spray.Seed® or paraquat is to kill plants that survived the glyphosate. Reducing the rate of the second knock risks survival of potentially glyphosate resistant individuals and damages the integrity of the double knock tactic. Remember that paraquat and Spray.Seed® are contact herbicides and require robust water rates to ensure adequate coverage, and allow for losses on stubble.

If the main weed problem is annual ryegrass then using paraquat on its own as the second knock is an appropriate choice. If there are also broadleaf weeds present, then the paraquat + diquat combination (e.g. Spray.Seed®) will be more effective overall. Mixing the glyphosate and paraquat together is both ineffective and not registered. Applying the two sprays between one and seven days apart is optimum timing.

If there is a mix of weeds present it can be useful to include a compatible herbicide 'spike' such as 2-4D low volatile ester, carfentrazone, or oxyflouren to enhance control of broadleaf weeds. Be very mindful of plant-back requirements of some herbicide 'spikes' before planting sensitive crops such as pulses and canola.

Don't rely on a pre-sowing double knock alone. Use pre-emergent herbicides, and focus on increasing the level of crop competition with narrow row spacing and varieties with vigorous early growth. Sow cereals at the optimal time to maximise competitiveness.

In weedy paddocks, consider the value of break crops such as pulses, canola, or hay as a way of incorporating other in-crop and non-chemical options to manage annual ryegrass, such as grass-selective post-emergent herbicides, crop-topping, desiccation, spraying under the swath or narrow windrow burning where appropriate. [19]

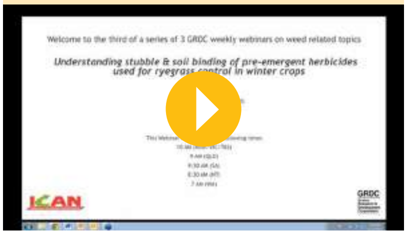
18 GRDC (2014). Summer fallow weed management. https://grdc.com.au/Resources/Publications/2014/05/Summer-fallow-weed-management

19 WeedSmart. 2015. How can I get the best best for my buck with a double knock? http://www.weedsmart.org.au/ask-an-expert/how-can-i-get-the-best-bang-for-my-buck-with-a-double-knock/

VIDEOS

4. Pre-emergent herbicides – Pt 1 Solubility and Binding.

5. Webinar - Stubble & soil binding of pre-emergent herbicides for annual ryegrass control in winter crops.



6.7 Pre-emergent herbicides

Pre-emergent herbicides are applied to the soil either before or directly after sowing and prior to weed emergence.

The pre-emergent herbicides alone will not adequately control large weed populations, and so they need to be used in conjunction with paddock selection and pre-seeding weed control.

Selection of the appropriate pre-emergent herbicide can only be made after assessing such factors as weed spectrum, soil type, farming system, and local experience. Refer to the complete product label for directions for use, application rates, weeds controlled, and conditions for best results.

Pre-sowing application is possible with some products and is often safer than post-sowing application because the sowing operation removes a certain amount of the herbicide from the crop row. Higher rates can often be used pre-sowing, but in both cases, the rate must be adjusted to soil type, as recommended on the product label.

6.7.1 Why use pre-emergent herbicides?

Pre-emergent herbicides are an essential part of a conservation farming system for a number of reasons:

- They can offer alternative modes of action to post-emergent herbicides.
- Many are very effective on hard-to-kill weeds such as annual ryegrass and barley grass.
- The current level of herbicide resistance to pre-emergent herbicides in NSW is very low.
- Pre-emergent herbicides control weeds early in crop life and potentially over multiple germinations, maximising crop yield potential.
- They suit a no till seeding system with knife points and press wheels and/or disc seeders.
- They can be cost effective. [20]
- There is also limited options for post emergent weed control in pulses. [21]

Whilst pre-emergent herbicides can be used in conservation farming systems, they must be used in conjunction with herbicide/crop rotation management plans and other non-chemical weed control techniques (Photo 4). These methods usually aim to minimise weed seed production and may include fallows, crop rotations including pastures and/or cutting hay, burning full paddocks or windrows, chaff carts, and weed seed destructors. [22]

Photo 4: *Trials have identified pre-emergent product combinations that provided significantly better control (right) than the district standard practice (left) for herbicide resistant annual ryegrass.*

Source: WeedSmart.

20 Haskins, B. NSW DPI. Using pre-emergent herbicides in conservation farming systems.

21 Stuchbery J. (2016) Personal communication.

22 Haskins, B. NSW DPI. Using pre-emergent herbicides in conservation farming systems.

VIDEOS

6. Webinar - Understanding pre-emergent herbicides.

6.7.2 Herbicide options

Chickpeas are late maturing (compared to other pulses), hence crop-topping to prevent ryegrass and other weed seed set is not possible, even in the earliest of maturing varieties (e.g. Genesis 079). Chickpeas are relatively slow to emerge with slow early growth during the colder winter months. As a consequence, they are poor competitors with weeds. Even moderate weed infestations can cause large yield losses and harvest problems.

The weed control strategy for growing a successful chickpea crop depends on substantially reducing the viable weed seed bank in the soil before the crop emerges. Control the majority of weeds before seeding, either by cultivation or with knockdown herbicides such as glyphosate or Spray Seed®.

A technique used with varying success by growers has been to sow chickpea and then use a knockdown herbicide tank mixed with a pre-emergent herbicide to control germinating weeds before the crop emerges. Chickpea crops may take up to 21 days to emerge under cool, drying soil conditions but under favourable warm, moist soil conditions plants may emerge after seven days. Done well, this can be an effective weed control option.

The pre-emergent herbicides will not adequately control large weed populations by themselves, and so they need to be used in conjunction with paddock selection and pre-seeding weed control. Incorporation by sowing (IBS) is generally considered safer on the crop than post-sowing pre-emergence with most herbicides used in modern no till sowing systems. Most of these products work best if thoroughly incorporated with soil either mechanically or by irrigation or rainfall. The aim of incorporation is to produce an even band of herbicide to intercept germinating weed seeds.

Simazine is the most widely used herbicide for broadleaf weed control, and can provide relatively cheap control of cruciferous weeds. Efficacy is very dependent on receiving rainfall (20–30 mm) within 2–3 weeks of application, and consequently weed control is often disappointing under drier conditions.

Balance® (isoxaflutole) is a systemic herbicide belonging to the relatively new class of isoxazole herbicides (Group H). Balance provides more consistent and reliable control of susceptible weeds for longer and across a broader range of seasonal conditions.

Terbyne is the newest triazine herbicide to be introduced in Australia and is registered for pre-emergent weed control in chickpeas, lupin, field peas, faba beans, lentils and triazine tolerant canola. Terbyne can be applied pre or post sowing.

Terbyne controls a wide range of broadleaf weeds, with some suppression of grasses, particularly if there is good soil moisture. Sufficient rainfall (20–30 mm) to wet the soil through the weed root zone is necessary within 2–3 weeks of application.

Spinnaker® (700 g/kg imazethapyr). For the pre-or post-emergence control of certain weeds in Centrosema (Cavalcade), chickpeas, faba beans, field peas, lucerne, mung beans, peanuts, serradella, soybeans and subterranean clover as per the directions for use table.

In chickpea crops sown on wide rows, there is increasing adoption of 'directed sprays' of Broadstrike, either alone or in tank-mixes with simazine. This largely avoids the problem of crop damage and improves weed control through the ability to safely add wetters or mineral oils to the spray mix.

Directed sprayers are most common in or around the cotton growing areas, as they enable relatively cheap grass and broadleaf weed control using glyphosate in-crop. While chickpeas do have a degree of tolerance to glyphosate during the vegetative stage, caution is still required as the lower branches arising from the main stem contribute a large proportion of the total chickpea yield. Upright varieties such as Amethyst and Jimbour are more suited to this technique than the more prostrate types and small chickpea plants are more susceptible to damage than older plants. [23]

23 Pulses Australia. Chickpea Production: Southern and Western regions. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

Balance® (isoxaflutole) is a Group H herbicide and its use is specific to chickpeas for broadleaf weed control in broadacre cropping situations. It provides a weed control option unique to chickpeas and enables rotation of herbicide groups across the cropping sequence.

Pre-emergent herbicides in the high-rainfall zone

The seasonal conditions that unfold can have a significant effect on the efficacy of any product applied. Pre-emergent herbicide performance depends on herbicide characteristics, solubility, affinity to soil, modes of degradation and the weed spectrum targeted, germination characteristics, soil/sowing conditions, expected moisture/environmental conditions and the complex interaction of these factors. For example, some products are not well-suited to higher rainfall zones because multiple weed germinations are more likely and by later in the season the herbicide has dissipated or moved too far down the soil profile to have any effect on later germinations. [24]

An early series of trials were conducted to evaluate the selectivity of a range of herbicides in chickpeas (Table 16). Trifluralin had a narrow safety margin and rates in excess of 0.56 kg/ha were damaging. Pendimethalin produced similar damage at rate of 0.99 kg/ha and 1.98 kg/ha to trifluralin at rate of 0.84 kg/ha and 1.12 kg/ha. These herbicides reduced plant establishment by 15–38% with the greatest reduction at the higher rates. Where trifluralin-reduced chickpea stands to below 40 plants/m^2, a significant yield reduction of 14.5% occurred. The wild oat herbicide triallate, was safe on chickpeas at rate up to 2.24 kg/ha. When trifluralin was added to triallate, damage tended to be slightly worse than with trifluralin alone. [25]

Table 16: *Tolerance of chickpeas to pre-plant incorporation of herbicides.* [26]

Herbicide	Rate (kg a.i./ha)	No. of experiments (all harvested)	Significant reductions*	Average Yield (s.e.) (% site maximum)	Tolerance rating
Tri-allate	0.56-0.8	2	0	90.5 92.1)	T
	1.12-2.24	2	0	89.5 (9.2)	T
Pendimethalin	0.99	1	0	100	MT
	1.98	1	0	79	MS
Trifluralin	0.56	4	0	89.8 (5.0)	T-MT
	0.84	2	0	87.5 (4.9)	MT-MS
	1.12	2	1	85.5 (9.2)	MS
Trifluralin + Tri-allate	0.56+0.56	2	0	84.5 (12.0)	MT
Cyanazine+Trifluralin	2.0+0.40	1	0	89	T
Prodiamine	0.398	1	0	100	T
Unweeded control	-	4	1	86.3 (8.4)	
Handweeded control	-	3	0	96.3 (3.2)	-

* Number of experiments where yield < site maximum (P = 0.05).

6.7.3 Application

Most products work best if incorporated into soil, either mechanically or by irrigation or rainfall. The aim of incorporation is to produce an even band of herbicide to intercept germinating weed seeds. Some herbicide incorporation occurs when sowing is done with knife-points, provided sowing speed is adequate to throw soil

24 WeedSmart. Gearing up to use pre-emergent herbicides. http://www.weedsmart.org.au/webinars/setting-crops-up-for-success/

25 G Kay, M, McMillan. (1990). Pre and post emergent herbicides in chickpeas I. Crop tolerance. Proceedings of the 9th Australia Weed Conference.

26 G Kay, M, McMillan. (1990). Pre and post emergent herbicides in chickpeas I. Crop tolerance. Proceedings of the 9th Australia Weed Conference.

VIDEOS

7. GCTV16: Spray application Workshop.

into the inter-row without throwing into the adjacent seed furrow. Hence, these products are still compatible with the shift to minimum tillage and reduced-tillage farming practices. However, there may be insufficient soil throw with some low-disturbance, disc seeding systems. Typically, a follow-up, post-emergent grass weed herbicide is still required to provide the level of grass weed control desired by growers, particularly in the seed furrow.

6.7.4 Herbicide efficacy in retained stubble systems

The GRDC project 'Maintaining profitable farming systems with retained stubble—upper Eyre Peninsula' aims to improve farm profitability while retaining stubble in farming systems on upper Eyre Peninsula (EP). One of the barriers to retaining stubble is the perceived reduction in pre-emergent herbicide effectiveness in stubbles. This component of the project is testing whether various stubble management activities impact on herbicide efficacy.

Weed control in stubble-retained systems can be compromised when stubbles and organic residues intercept the herbicide and prevent it from reaching the desired target, or when the herbicide is tightly bound to the organic matter. Reduced herbicide efficacy in the presence of higher stubble loads is a particular issue for pre-emergent herbicides. Current farming practices have also changed weed behaviour with a shift in dormancy in barley grass genotypes now confirmed in many paddocks of the Minnipa Agricultural Centre.

To measure the efficacy of herbicides in different stubble management systems, two different stubble management strategies were implemented at harvest: traditional spread stubble and harvest windrows. The third stubble management strategy was total stubble removal by burning. The harvest windrows within the trial area were also burnt. Measurements taken were stubble load pre-seeding, soil moisture, plant emergence count, early and late grass weed count, medic growth score, grain yield, and grain quality.

Stubble treatments (averaged over all chemical treatments)

Early dry matter and grain yield were lower in the spread stubble system than the burnt stubble and burnt windrow systems and this may have been due to less moisture reaching the seedbed and also the tie up of nitrogen resulting in early nitrogen deficiency (Table 17). There may also have been some yellow leaf spot interactions.

Table 17: *Effect of cereal stubble management on crop establishment, dry matter and yield as well as weed and medic populations in 2015.*

	Establishment (plants/m²)	Early crop dry matter (t/ha)	Early in-crop barley grass 24 July (plants/m²)	Medic growth (0–3 rating)*	Late in-crop barley grass 26 Oct (plants/m²)	Yield (t/ha)
Burnt stubble	105	0.22	3.1	1.01	6.8	1.63
Spread stubble	93	0.19	1.8	0.78	4.8	1.55
Burnt windrows	97	0.22	6.7	0.94	10.3	1.69
LSD (P=0.05)	4	0.02	1.7	0.12	2.7	0.04

* Visual medic rating system where 0=no medic, 1=small suppressed medic, 2=small and large medic, 3=mostly large medic plants

Source: GRDC.

Chemical treatments

There were no impacts of stubble management on the performance of individual chemical treatments so results presented in this section are averaged over all three stubble management treatments.

Trifluralin and Diuron mixes caused some crop damage but the crop recovered better than expected and dry matter production of the crop was as good as in the untreated control by sampling time, this was probably due to less soil water movement of the chemicals. In a dry start, Boxer Gold® did not appear as effective on barley grass as ryegrass, but post-application gave some suppression activity on all grasses.

Despite high cereal stubble loads, completely removing stubble by burning did not improve the efficacy of any of the chemical packages tested in this trial. The generally low grass weed pressure observed in this trial, however, makes it difficult to draw any conclusive recommendations relating to the impact of stubble management based on these results alone.

When choosing the most appropriate pre-emergent herbicide for use in stubble retained systems, it is important to consider:

- the likely rainfall pattern and soil moisture conditions post application;
- the susceptibility of the crop to the herbicide;
- the position of the weed and crop seeds in the soil profile;
- the mobility of the herbicide in soil water;
- the persistence of the herbicide activity relative to the germination pattern of the target weeds;
- specific tillage/seeding system and level of soil disturbance, and;
- long-term objectives of the weed management program. [27]

MORE INFORMATION

APVMA

Using pre-emergent herbicides in conservation farming

Pre-emergent herbicides Factsheet.

6.8 Post-plant pre-emergent herbicides

When a pre-emergent herbicide is applied after sowing (but before crop emergence) to the seedbed (Table 18).

These herbicides are primarily absorbed through the roots, but there may also be some foliar absorption (e.g. Terbyne®). When applied to soil, best control is achieved when the soil is flat and relatively free of clods and trash. Sufficient rainfall (20–30 mm) to wet the soil through the weed root-zone is necessary within 2–3 weeks of application. If applied pre-sowing and sown with minimal disturbance, incorporation will essentially be by rainfall after application. Weed control in the sowing row may be less effective because a certain amount of herbicide will be removed from the crop row.

The absence of cost-effective and safe post-emergent herbicides effectively limits broadleaf weed control options in chickpeas to a small number of pre-emergent herbicides. Most of these chemicals are very dependent on rainfall soon after application, and as a consequence often result in inconsistent or partial weed control under drier conditions. [28]

An early series of trials were conducted to evaluate the selectivity of a range of herbicides in chickpeas (Table 18). The herbicides which performed best under post-plant pattern were all triazines. In one experiment, mixtures with simazine were more dmaaging than herbicides applied alone at comparable total rate of the active triazine. Chickpeas have consistently shown a haigh tolerane to registered herbicide cyanazine at rates up to 3.0 kg/ha and to prometryn at rates of 2.0 kg/ha or less. Damage symptoms were temporary and did not affect yields. Metribuzin was usually safe at a rate of 0.21 kg/ha but in one weedfree experiment a significant yield reduction occurred at this rate.

Table 18: *Tolerance of chickpeas to post-sowing, pre-emergence herbicides.* [29]

27 Cook A, Bates A, Shepperd W, Richter I. GRDC Update Papers (2016). Herbicide efficacy in retained stubble systems. https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/08/Herbicide-efficact-in-retained-stubble-systems

28 Pulses Australia. Chickpea Production: Southern and Western regions. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

29 G Kay, M, McMillan. (1990). Pre and post emergent herbicides in chickpeas I. Crop tolerance. Proceedings of the 9th Australia Weed Conference.

Herbicide	Rate (kg a.i./ha)	No. of experiments (all harvested)	Significant reductions*	Average Yield (s.e) (% site maximum)	Tolerance rating
Acifluorfen	0.224-0.896	7 (7)	0	93.8 (5.4)	T-MT
Atrazine	1.0	3 (3)	2	49.0 (40.0)	T
Cynazine	1.5	9 (8)	0	4.9 (4.9)	T
	2.0	5 (4)	0	88.0 (1.6)	T-MT
	3.0	1 (1)	0	98	T-MT
Dimethazone	0.9	1 (1)	1	59	HS
Imazaquin	0.3	1 (1)	1	41	HS
Imazethapyr	0.05-0.07	2 (2)	2	77.5 (3.5)	S
Linuron	1.5	1 (1)	0	92	MT
Methabenzthiazuron	1.75	2 (1)	0	94	T-MT
Metribuzin	0.21	10 (8)	2	88.9 (10.6)	T-MT
	0.28	3 (3)	3	61.0 (26.2)	MT-MS
	0.42	1 (1)	1	70	MS
Prometryn	0.75	3 (3)	0	95.3 (4.0)	T
	1.5	9 (8)	1	92.8 (5.8)	T
	2.0	2 (2)	0	94.5 (7.8)	T-MT
	3.0	1 (1)	0	87	MS
Simazine	0.75	6 (5)	1	87.8 (6.3)	T
	1.0	3 (3)	1	88.7 (1.2)	T
	1.25	1 (1)	1	85	T-MT
	1.5	6 (5)	2	87.0 (8.5)	T-MT
	2.5	1 (1)	1	55	MS
Terbutryn	2.0	2 (2)	1	78 (2.8)	T-MT
Cyanazine + metolachlor	2.0 + 1.44	1 (1)	0	82	T
Cyanazine + simazine	0.75-1.0+0.75	6 (5)	1	89.4 (8.7)	T-MT
Metribuzin + simazine	0.105-0.14+0.75	6 (5)	0	91.6 (6.2)	MT
Prometryn + simazine	0.75+0.75	6 (5)	1	91 (6.5)	T-MT
	1.5+0.75	3 (3)	0	89 (4.2)	MT
Unweeded control	-	13 (12)	5	76.9 (23.0)	-
Handweeded control	-	10 (9)	0	94.6 (4.7)	-

* Number of experiments where yield < site maximum (P = 0.05).

Results with simazine suggest that chickpeas were tolerant to rates less than 1 kg/ha (of active ingredient) but significant yield reductions due to poor weed control occurred at these rates in two experiments. Some variation was found on different soil types. In one trial on grey clay soils chickpea yield was reduced by 0.4 t/ha in the simazine 1.25 kg/ha treatment but on other soil types, no yield reduction occurred at 1.5 kg/ha. Damage symptoms developed slowly and usually increased as the season progressed. Yield losses from simazine were greater than would be expected from visual damage ratings. [30]

30 G Kay, M, McMillan. (1990). Pre and post emergent herbicides in chickpeas I. Crop tolerance. Proceedings of the 9th Australia Weed Conference.

IN FOCUS

Effect of row spacing, nitrogen and weed control on crop and weed in a wheat – lupin or wheat – chickpea rotation

Key messages

- Despite a very dry season in 2012, dimethenamid (e.g. Outlook®) herbicide in a lupin or chickpea crop was more effective on annual ryegrass than simazine in the two long-term rotational trials at Cunderdin and Merredin. This has resulted in an increase in grain yield of lupin crop in Outlook® treatment at Cunderdin.
- Sakura® reduced annual ryegrass head numbers more effectively than trifluralin at $N_2$5 and flexi N50 compared to N50 at Merredin.
- Grain yields of both crops at Merredin were very poor. Despite poor grain yields of crops at Merredin, yields of both crops were greater at 44 cm row spacing than at 22 cm row spacing.

In 2012, rotation trials of three years duration were initiated at Cunderdin (wheat–lupin) and at Merredin (wheat– chickpea) to examine the effect of crop row spacing, herbicides and applied nitrogen (in wheat only) on crops and weeds.

Conclusions

Rainfall was extremely low at both sites in the 2012 season leading to very poor crop growth. Dimethenamid (e.g. Outlook®) herbicide was more effective on annual ryegrass than simazine in lupin and chickpea crops resulting in greater lupin grain yield. Even though grain yields of crops were very low, yields of both crops at Merredin were greater at 44 cm row spacing than at 22 cm row spacing. These results showed the benefit of wide row spacing in a dry season like 2012 in low rainfall areas. However, under high weed competition at Cunderdin, narrow row spacing appeared more productive with Outlook® herbicide than wide row spacing. [31]

Chickpea post-emergent herbicide trials 2014

Take home messages

- Factor® (butroxydim) was not effective as a knockdown for wild oats in chickpeas.
- Mixtures of Verdict® (haloxyfop) plus Status® (clethodim) or 500 mL/ha of Status® proved the most effective brews for knockdown control of wild oats.
- Clethodim (as Status®) benefited from the addition of Liase® even when rain water was used as the carrier. Be sure to follow label conditions.
- In reviews of the 2013 winter season agronomists and farmers had raised managing resistant populations of wild oats as a major issue for the Northern region and an emerging issue for the Southern region. Wild oats are a key weed of winter crops throughout the region and for the past 30 years growers and agronomists have relied on Group A herbicides for control.

Haloxyfop, a “fop” herbicide, is perhaps the most widely used herbicide in the Northern region for knockdown of wild oats in chickpeas and other pulse crops, however resistance to this herbicide is becoming increasingly frequent. This could

31 A Hashem, W Vance, R Brennan, R Bell. (2013). DAFWA 2013 Crop Updates. Effect of row spacing, nitrogen and weed control on crop and weed in a wheat – lupin or wheat – chickpea rotation.

eventually threaten Southern crops as well. Many agronomists, where they have concerns about the Group A resistance status of wild oats in a paddock, will mix haloxyfop (e.g. Verdict 520) with a "dim" herbicide (most commonly clethodim), other agronomists might use clethodim alone.

Butroxydim (e.g. Factor) is another "dim" group A that although registered for wild oat control in many broadleaf crops has not been widely used in NSW.

MORE INFORMATION

Wide rows and stubble management.

6.9 In-crop herbicides: knock downs and residuals

Chickpeas can be grown in wider rows in a stubble system that allows inter-row herbicide application with shielded sprayers.

Problem weeds or situations that require special attention include:

- Group A ('dims' and 'fops') resistant wild oats (and other grass species)
- late germinations of weeds (e.g. ryegrass, brome grass) that would normally be prevented from setting seed in other pulses through croptopping
- snail medic, which can escape Balance®
- hoary cress, soursob, tares, wild vetch, bedstraw, bifora, muskweed, wild radish and volunteer pulses[32]

Broadstrike® usually causes some transient crop yellowing and can cause reddish discoloration and height suppression. Flowering may be delayed (Photo 5), resulting in yield suppression.

Photo 5: *To control turnip weed, a single boom width of Broadstrike® was applied. Flowering and maturity of treated chickpeas (left) was delayed significantly, so they are still green compared with the untreated chickpeas that have matured (right).*

Photo: G. Cumming, Pulse Australia.

Broadstrike® is used mainly in salvage situations (as a last resort), and even then should be applied only under good growing conditions. Photo 6 depicts effective use of Broadstrike® against turnip weed adjacent to a chickpea crop.

With the shift into row-crop chickpeas, some growers are successfully using Broadstrike® as a directed spray into the inter-row area. This keeps a large proportion of the herbicide off the chickpea foliage and minimises crop damage.

32 Pulses Australia. Chickpea Production: Southern and Western regions. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

Photo 6: *The same single boom width of Broadstrike® applied along the chickpea crop edge (centre) alongside the unsown, weedy headland (right) and untreated crop (far left). Broadstrike® did an excellent job on the turnip weed (centre and unsown front) compared with the untreated headland (right).*

Photo: G. Cumming, Pulse Australia.

6.9.1 Directed sprays

Though row-cropping chickpeas on wide rows in the Southern region is not commonly practiced, it provides an opportunity for the use of 'directed sprays' of Broadstrike®, either alone or in tank-mixes with simazine. This largely avoids crop damage and improves weed control through the ability to add wetters or mineral oils safely to the spray mix.

6.9.2 Shielded sprayers

Although chickpeas do have a degree of tolerance to glyphosate during the vegetative stage, caution is still required as the lower branches arising from the main stem make a large contribution to the total chickpea yield. Issues that need to be considered include:

- selection and operation of spray shields (speed, nozzle type, etc.)
- height of the crop (small chickpea plants are more susceptible)
- variety (upright types are more suited to this technique than the more prostrate types)

6.9.3 Brome and barley grass management in cropping systems of southern Australia

Take-home messages:

- Increasing incidence of brome and barley grass in cropping paddocks in southern Australia is likely to be associated with selection of more dormant biotypes by weed management practices used by the growers.
- At present brome grass management in cereals is heavily reliant on group B herbicides, especially the Clearfield™ technology. Delaying onset of resistance to these herbicides would require identification of effective alternative herbicides.
- Field trials undertaken over four years have investigated various pre-emergence herbicides for brome grass control in wheat. Even though Sakura® (pyroxasulfone) appears to be the most active pre-emergence herbicide against brome grass, it lacks consistency required for long-term population management of brome grass.

- Field trials over four years confirmed consistently high efficacy of Sakura® against barley grass, especially under situations with good soil moisture.
- Barley grass management is now being complicated by evolution of group A resistance in this weed species. However, there appear to be several effective alternatives (e.g. Sakura® and Raptor®) that could be used for barley grass control in broadleaf crops.

Feedback from growers and consultants in southern Australia has clearly shown increasing spread of brome and barley grass.

Brome grass management in broadleaf crops is heavily reliant on the use of group A herbicides. However, there are more than 30 cases of confirmed resistance in brome grass to group A herbicides.

Herbicide rotation has been widely recognised as being important to delay onset of resistance to herbicides. In order to implement herbicide rotations to delay group B resistance in brome grass, it is important to identify alternative modes of action.

Field studies have been undertaken over four years to investigate the performance of alternative modes of action to control brome grass. Sakura® and its combination with other herbicides were investigated for the control of brome grass. In some of these field trials, Sakura® alone provided excellent control (>90%) of brome grass (Figure 1). However, at the other trial sites, the level of weed control was quite disappointing (25%). At this stage, underlying reasons for this large variability in the performance of Sakura® on brome grass are unclear. Use of split application of Sakura® was equally variable as its single pre-sowing application. Addition of Avadex® (triallate) to Sakura® improved brome grass control relative to Sakura® alone in 2012 but the level of weed control obtained at Balaklava was inadequate (<60%). However, this treatment appears to be worthy of further research. In most situations, addition of trifluralin to Sakura® did not significantly improve brome grass control. In summary, it could be argued that none of the currently available pre-emergence herbicides have the required stability in efficacy to become viable alternatives to post-emergence group A or imidazolinone herbicides.

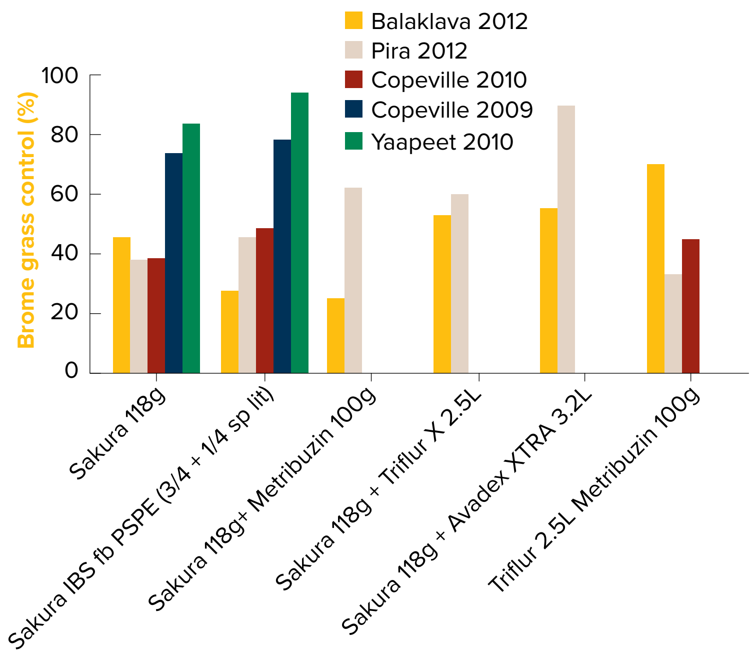


Figure 1: *Effect of different pre-emergence herbicides on the control of brome grass in field trials. It should be noted that rates of Triflur X and Avadex® were lower in trials undertaken prior to 2012. Weed control is expressed as reduction in brome grass plant density.*

Source: GRDC.

Barley grass management

Many growers have reported an increasing incidence of barley grass in their crops. Weed management practices used in cropping systems have selected for increased seed dormancy, which is likely to contribute to greater abundance of this weed species in field crops.

Release of pyroxasulfone (Sakura®) in Australia has been an important development in the management of barley grass. In many field trials undertaken on the Eyre Peninsula over four years, Sakura® consistently provided effective control of barley grass (Figure 2). Unfortunately, many farmers are still using cheaper but inferior herbicide options for barley grass, which can lead to large build-up in weed infestations.

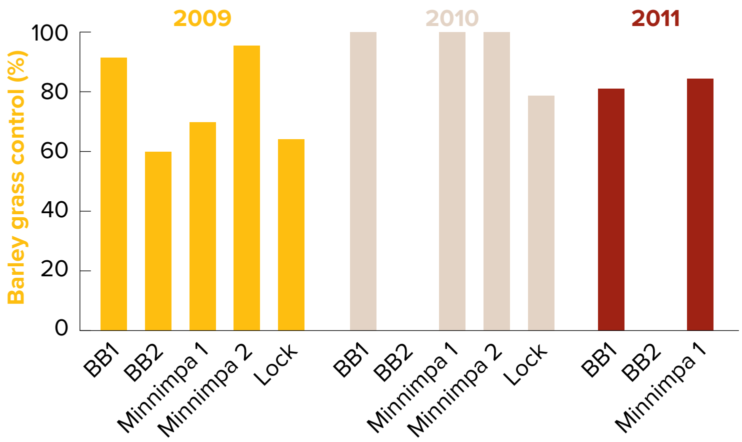


Figure 2: *Effect of Sakura® at the recommended rate (118 g/ha) on barley grass control in wheat at trial sites on the Eyre Peninsula. Weed control is expressed as reduction in barley grass seed production. BB = Buckleboo; 1 and 2 represent time of sowing.*

Source: GRDC.

Presence of high levels of resistance to group A herbicides is a major concern for weed management in pulse crops. In order to investigate the performance of alternative herbicides on group A resistant barley grass, a field trial was conducted at Baroota in 2012. Sakura®, Raptor® (imazamox), and an experimental compound provided excellent control of barley grass, which was reflected in significant increases in grain yield of field peas (Table 19). Outlook® (dimethenamid) appeared to be relatively ineffective early in the season but its performance improved with time and it may have a useful role in field peas. [33]

33 GRDC Update Papers. 2013. Brome and Barley grass management in cropping systems of southern Australia. https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/02/Brome-and-barley-grass-management-in-cropping-systems-of-southern-Australia

Table 19: *Effect of different herbicide treatments on grain yield of field peas and reduction in group A resistant barley grass seed production at Baroota (SA) in 2012.*

Treatments	Seed set reduction (%)	Field pea yield (t/ha)
Sakura® @ 118 g/ha IBS	99	2.29
Boxer Gold® @ 2.5 L/ha IBS	74	1.41
Outlook® @ 1 L/ha IBS	93	2.14
Raptor® @ 45 g/ha + BS1000 0.2% PE	100	2.08
Trifluralin @ 2.0 L/ha + Avadex® Xtra @ 2.0 L/ha	71	1.32
Metribuzin @ 200 g/ha PSPE	46	0.82
Propyzamide 500 g a.i./ha	100	2.29
Diuron 900@ 1 kg/ha + Trifluralin @ 2.0 L/ha IBS	78	1.58
Trifluralin 2.0 L/ha IBS	68	1.19
Control	-	0.82
LSD (P=0.05)		0.33

Source: GRDC.

6.10 Conditions for spraying

All grass herbicides labels emphasise the importance of spraying only when the weeds are actively growing under mild, favourable conditions (Photo 7). Any of the following stress conditions can significantly impair both uptake and translocation of the herbicide within the plant, likely resulting in incomplete kill or only suppression of weeds:

- moisture stress (and drought)
- waterlogging
- high temperature–low humidity conditions
- extreme cold or frosts
- nutrient deficiency, especially effects of low nitrogen
- use of pre-emergent herbicides that affect growth and root development; i.e. simazine, Balance®, trifluralin, and Stomp®
- excessively heavy dews resulting in poor spray retentions on grass leaves

Photo 7: *Boom spray on crop.*

Photo: Brad Collis, Source: GRDC.

Ensure that grass weeds have fully recovered before applying grass herbicides.

Group A herbicides can occasionally cause leaf spotting in chickpeas (Photo 8). This is usually associated with either frost or high temperatures occurring soon after spray application. [34]

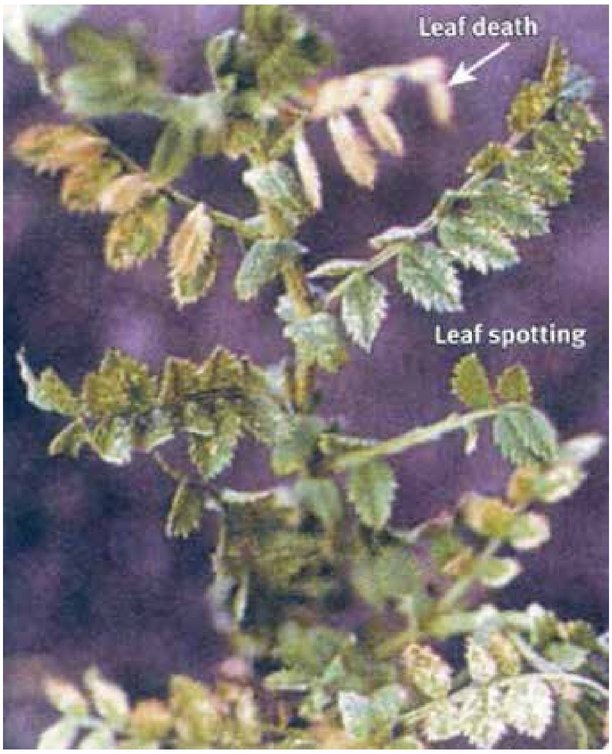


Photo 8: *Group A grass selective herbicide injury.*

Photo: T. Bretag.

VIDEOS

8. Advances in weed management – Webinar 2 – Spray application in summer fallows.

34 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

MORE INFORMATION

Cultivar herbicide tolerance trial protocols

6.11 Herbicide tolerance ratings, NVT

Within many broad acre crop species, cultivars have been found to vary in sensitivity to commonly used herbicides and tank mixes, thereby resulting in potential grain yield loss, and hence reduced farm profit. With funding from GRDC and State Government Agencies across Australia, a series of cultivar by herbicide tolerance trials are conducted annually. The trials aim to provide grain growers and advisers with information on cultivar sensitivity to commonly used in-crop herbicides and tank mixes for a range of crop species including chickpeas, lupins, peas, lentils and faba beans. The intention is to provide data from at least two years of testing at the time of wide scale commercial propagation of a new cultivar. [35] See the results of the most recent trials below.

Pulse variety response to herbicides in Victoria

Pulse variety response to herbicides in South Australia

6.11.1 Developing improved herbicide tolerance in pulse crops

Herbicides are the main method of weed control in broadacre intensive farming systems and the development of herbicide tolerance traits in pulse crops has been identified as a major breeding priority. The recent release and rapid adoption of the first herbicide tolerant lentil XT varieties demonstrates the likely demand for these traits in other pulse crops, particularly faba beans where no in-crop broadleaf weed control options are currently available. Additionally, the development of multiple herbicide tolerances, particularly for different modes of action, is important to ensure robust and sustainable weed control options into the future. This project explored a number of different strategies to develop lines with improved tolerance to key herbicides in chickpeas, faba beans, and lentils.

Mutagenesis methods have been successfully used in the development of novel herbicide tolerance traits in a number of commercialized crops. In this project, mutagenized populations of lentils, faba beans, and chickpeas were screened for tolerance to a range of herbicides, and selections with high levels of putative tolerance were identified in each crop (Table 20). Details of the herbicide tolerant traits for each crop are described below.

Table 20: *Summary of herbicide tolerant germplasm developed through mutagenesis methods.*

	Chickpea
Mutated cultivar	PBA HatTrick$^{\phi}$
Herbicide	clopyralid
Year/s screened	2014
Population size screened	Five million M_2 seeds
Field selections collected	67 M_2 plants
Lines progressed with herbicide tolerance trait	50 lines
Level of improved tolerance developed	High
Current status of validation	Dose response experiments 2016

Source: GRDC.

Fifty chickpea lines were confirmed to have improved tolerance to clopyralid in progeny screen experiments, with preliminary experiments indicating a high level of tolerance in these lines (Photo 9).

35 Ramsey C, Wheeler R, Churchett J, Walker S, Lockley P, Dhammu H, Garlinge J. (2010). Cultivar herbicide tolerance trial protocols. http://www.nvtonline.com.au/wp-content/uploads/2013/02/Herbicide-Tolerance-Protocols.pdf

All lines were bulked up during the 2015 season to multiply seed for future studies including dose experiments in 2016, and PBA Chickpea has started opportunistically incorporating lines with this trait into the breeding program.

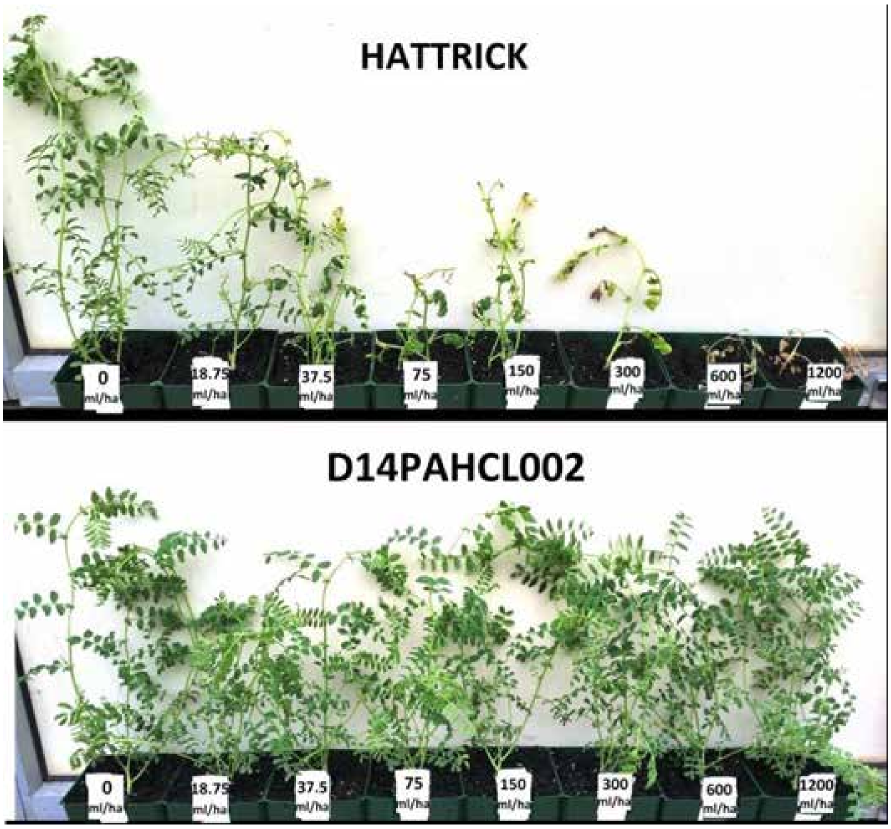


Photo 9: *Photo from a preliminary clopyralid dose response showing improved tolerance levels of a chickpea selection D14PAHCL002 compared to control cultivar PBA HatTrick*⊕.

Source: GRDC.

Summary and future work

The development of lines with low levels of herbicide tolerance from existing germplasm as well as high levels of improved tolerance from novel germplasm will help to improve grower confidence, expand weed control options and reduce the rotational limitations of pulse crops. All traits are being progressed in PBA breeding programs and new traits will continue to be evaluated in dose response and field trials as seed becomes available. Molecular markers will continue to be developed for all traits wherever possible; however, this may be difficult in selections from existing germplasm with low levels of tolerance as they are likely to be complex (multi-gene) traits. Selections from novel germplasm can potentially carry deleterious genes and further work may be required to understand any limitations associated with these new traits. Further characterisation of these traits, such as evaluation of tolerance levels to other herbicides with the same mode of action, is also required to allow the best registration opportunities to be pursued. Additionally, future work in developing tolerance to different herbicides with different modes of action is also necessary in lentils and faba beans, and could also be extended to other crops such as field peas, to ensure robust and sustainable weed control options into the future. [36]

36 GRDC Update Papers (2016). Developing improved herbicide tolerance in pulse crops. https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/02/Developing-improved-herbicide-tolerance-in-pulse-crops

6.11.2 New opportunities for pulses in the Mallee

Take home messages

- Weed management options in pulses continues to improve through incorporation of multiple techniques in crop management including: in-crop weed control with herbicides, plant traits for enhanced weed competitiveness and reducing weed seed set. A particular highlight recently has been the release of the XT lentil varieties which have improved tolerance to Group B herbicides.
- Where Group B residues are not an issue or requirements for in crop application of imazethapyr are not required, careful consideration should be made to the yield and other trait benefits of several other conventional lentils which may prove more profitable.

The opportunities for pulses in the Mallee continue to expand as we see improvements in farming systems and genetic advancements in traits that confer improved adaptation to Mallee conditions. Several PBA chickpea, field pea, lupin, faba bean and lentil lines already display improved adaptation to the low-medium rainfall zone (LRZ) having been developed in the run of drought years, particularly through the late 2000s. Current advanced PBA breeding lines will have even greater adaptation to the LRZ, encompassing traits such as novel tolerance to herbicides, different flowering times and durations, boron and salt tolerance and high relative yield under drought conditions. These traits, combined with optimised agronomic management in the specific environments, will enable further expansion of pulses as a profitable and successful component of a farming system in the Mallee.

Bright future for weed control in pulses

Building on the success of the lentil research, the SARDI pre-breeding project has developed Group B tolerant faba beans and Group C tolerant lentil germplasm. Agronomic field trials in SA in 2014 confirmed faba bean lines with tolerance to a range of Imi chemistries and early generation PBA yield trials have identified lines with good adaptation to southern Australia. Preliminary field validation trials also confirmed a very high level of metribuzin tolerance (10–20 times) in lentil germplasm. This material has now been 'crossed' with the Group B tolerant lines with the aim of developing dual herbicide tolerant (Group B+C) varieties. The project is now expanding into the development of novel herbicide tolerance in both Kabuli and Desi chickpea. [37]

VIDEOS

9. GCTV16: Pulse Herbicide Tolerance.

6.12 Monitoring

Monitoring of weed populations before and after any spraying is an important part of management.

- Keep accurate records.
- Monitor weed populations and record results of herbicide used.
- If herbicide resistance is suspected, prevent weed seed-set.
- If a herbicide does not work, find out why.
- Check that weed survival is not due to spraying error.
- Conduct your own paddock tests to confirm herbicide failure and determine which herbicides remain effective.
- Obtain a herbicide-resistance test on seed from suspected plants, testing for resistance to other herbicide (MOA) groups.
- Do not introduce or spread resistant weeds in contaminated grain or hay.

Regular monitoring is required to assess the effectiveness of weed management and the expected situation following weed removal or suppression. Without monitoring, we cannot assess the effectiveness of a management program or determine how it might be modified for improved results. Effective weed management begins with

37 GRDC Update Papers (2015). New opportunities for pulses in the Mallee. https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2015/07/New-opportunities-for-pulses-in-the-Mallee

monitoring weeds to assess current or potential threats to crop production, and to determine best methods and timing for control measures.

Regular monitoring and recording details of each paddock allows the grower to:

- spot critical stages of crop and weed development for timely cultivation or other intervention;
- identify the weed flora (species composition), which helps to determine best short- and long-term management strategies; and
- detect new invasive or aggressive weed species while the infestation is still localized and able to be eradicated.

Watch for critical aspects of the weed-crop interaction, such as:

- weed seed germination and seedling emergence
- weed growth sufficient to affect crops if left unchecked
- weed density, height, and cover relative to crop height, cover, and stage of growth
- weed impacts on crops, including harbouring pests, pathogens, or beneficial organisms; or modifying microclimate, air circulation, or soil conditions; as well as direct competition for light, nutrients, and moisture
- flowering, seed-set, or vegetative reproduction in weeds
- efficacy of cultivations and other weed management practices

Information gathered through regular and timely field monitoring helps growers to select the best tools and timing for weed-control tactics. Missing vital cues in weed and crop development can lead to costly efforts to rescue a crop, efforts that may not be fully effective. Good paddock scouting can help the grower to obtain the most effective weed control for the least fuel use, labour cost, chemical application, crop damage, and soil disturbance.

6.12.1 Tips for monitoring

To scout weeds, walk slowly through the paddock, examining any vegetation that was not planted. In larger paddocks, walk back and forth in a zigzag pattern to view all parts of the paddock, noting areas of particularly high or low weed infestation. Identify weeds with the help of a good weed guide or identification key for your region, and note the weed species that are most prominent or abundant. Observe how each major weed is distributed through the paddock. Are the weeds randomly scattered, clumped or concentrated in one part of the paddock?

Keep records in a field notebook. Prepare a page for each paddock or crop sown, and take simple notes of weed observations each time the paddock is monitored. Over time, your notes become a timeline of changes in the weed flora over the seasons and in response to crop rotations, cover crops, cultivations and other weed control practices. Many growers already maintain separate records for each paddock; weed observations (species, numbers, distribution, size) can be included with these.

When to scout, and what to look for in a new paddock or farm

When purchasing farmland, it is important to look at the weeds. Presence of highly aggressive or hard-to-kill weeds, intense weed pressure, stressed and nutrient-deficient weeds, or a weed flora indicative of low or unbalanced soil fertility, pH or salt may foretell problems that should be considered when deciding whether to buy or rent, or how much to offer.

During your first year or two on a new farm or paddock, study the weeds carefully throughout the season, and be sure to get correct identification of the 5–10 most common weeds.

Note the weeds that emerge, grow or reproduce at different times of the annual cropping cycle:

- over winter

- after primary tillage and during seedbed preparation
- after crop planting
- during crop growth and maturation
- after harvest
- over summer or during cover crop emergence and establishment

Questions to ask include:

- What are the main weed species present at different times of year?
- When does each weed species emerge, flower, and set seed?
- What paddocks or areas have the worst weed pressure? The least?

6.13 Potential herbicide damage effect

Pulse crops can be severely damaged by some herbicides whether as residues in soil, contaminants in spray equipment, spray drift onto the crop or by incorrect use of the herbicide.

Leaching

Some soil active herbicides used for weed control in pulses can damage crops where conditions favour greater activity and leaching.

Herbicides move more readily in soils with:

- low organic matter
- more sand, silt, or gravel.

Herbicide movement is much less in soils with higher organic matter and higher clay contents. Damage from leaching is also greater where herbicides are applied to dry, cloddy soils than to soils which have been rolled and which are moist on top from recent rainfall. The pH of a soil can also strongly influence the persistence of herbicides. Many labels have warnings about high pH (≥8.0) and the need to reduce application rates to avoid crop damage. Heavy rainfall following application may cause crop damage. This will be worse if the crop has been sown shallow (less than 3–5 cm), where there is light soil and where the soil surface is ridged. The soil surface should not be ridged as this can lead to herbicides being washed down and concentrated in the crop row. [38]

Whilst trifluralin is relatively immobile in the soil, Boxer Gold® may move from the point of placement, particularly in sandy soils prone to leaching. Thus care must be taken in soils with a higher leaching potential and where previous history has shown potential for damage from herbicides with a higher leaching index such as Dual Gold®, metribuzin and the triazine herbicides. [39]

Based on Table 21, Metribuzin leaches at almost three times the rate of simazine and seven times the rate of diuron. The relative tolerance of the crop type and variety will also affect crop damage from these herbicides. For example, lupins are more tolerant to simazine than are the other pulses. For more specific details on soil active herbicides and the risk of crop damage in your cropping situation seek advice from an experienced agronomist.

Herbicide residues can last for several years, especially in more alkaline soils and where there is little summer rainfall. The pulses emerge and grow normally for a few weeks and then start to show signs of stress. Leaves become off-colour, roots may be clubbed, plants stop growing, and eventually die. Refer to the labels for recommendations on plant-back periods for pulses following use of any herbicides.

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38 GRDC (2008). Grain Legume Handbook – Weed control.

39 Douglas A. (2008). Weeds Update, Western Australia.

Contamination of spray equipment

Traces of sulfonylurea herbicides (such as chlorsulfuron, metsulfuron, or triasulfuron) in spray equipment can cause severe damage to legumes when activated by some of the grass control herbicides (Photo 10). The risk of residue damage is greater in the presence of grass-selective herbicides. Always clean spray tanks and lines with chlorine, according to recommendations, after using sulfonylurea herbicides and before using these grass control herbicides. Traces of Affinity® (and many registered herbicides) can also damage pulse crops.

Decontaminate with alkali detergent.

Photo 10: *Hygiene between spraying operations is essential. After using herbicide make sure the boom spray is cleaned out with chlorine before starting on grass control in legumes. The effect, as shown above, is dramatic.*

Source: GRDC.

Spray Drift

Pulse crops can be severely damaged by some herbicide sprays, such as 2,4-D ester, drifting into the crop (Photo 11). This can happen when these sprays are applied nearby in very windy or still conditions, especially where there is an inversion layer of air on a cool morning. When using these herbicides spray when there is some wind—to mix the spray with the crop. Do not use excessively high spray pressure as this will produce too fine a spray, which is more likely to drift onto a neighbouring pulse crop. [40]

40 GRDC (2008). Grain Legume Handbook – Weed control.

Photo 11: *Severe metsulfuron-methyl damage in chickpea plants.*

Source: DAFQ in Crop IT.

6.13.1 Avoiding herbicide damage

Some herbicides can severely damage chickpea crops through residues in soil, contaminants in spray equipment, spray drift onto the crop or by incorrect use of the herbicide.

The importance of cleaning and decontaminating spray equipment before the application of herbicides cannot be over-stressed. Traces of sulfonylurea herbicides (such as chlorsulfuron, metsulfuron, or triasulfuron) in spray equipment can cause severe damage to chickpea and other legumes when activated by grass control herbicides.

Taking some general precautions can help to reduce the likelihood of crop damage with residual herbicide use at planting:

- Do not apply residual herbicides if rain is imminent.
- Maintain at least 7.5–10 cm soil coverage.
- Avoid leaving a furrow or depression above the seed that could allow water (and chemical) to concentrate around the seed or seedling.
- Avoid leaving an exposed, open slot over the seed with disc-openers and avoid a cloddy, rough tilth with tined-openers. [41]

6.13.2 Plant-back intervals

Plant-back periods are the obligatory times between the herbicide spraying date and safe planting date of a subsequent crop.

Some herbicides have a long residual. The residual is not the same as the half-life. Although the amount of chemical in the soil may break down rapidly to half the original amount, what remains can persist for long periods (e.g. sulfonylureas (chlorsulfuron)). This is shown in the Table 21 and 22 where known. Herbicides with long residuals can affect subsequent crops, especially if they are effective at low levels of active ingredient, such as the sulfonylureas. On labels, this will be shown by plant-back periods, which are usually listed under a separate plant-back heading or under the "Protection of crops etc." heading in the "General Instructions" section of the label.

MORE INFORMATION

Field crop herbicide injury: The Ute Guide

Chickpea Disorders: The Ute Guide

41 Pulses Australia. Chickpea Production: Southern and Western regions. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

Part of the management of herbicide resistance includes rotation of herbicide groups. Paddock history should be considered. Herbicide residues (e.g. sulfonyl urea, triazines etc.) may be an issue in some paddocks. Remember that plant-back periods begin after rainfall occurs. [42]

Table 21: *Residual persistence of common pre-emergent herbicides, and note residual persistence in broad-acre trials and paddock experiences.* [43]

Herbicide	Half-life (days)	Residual persistence and prolonged weed control
Logran® (triasulfuron)	19	High. Persists longer in high pH soils. Weed control commonly drops off within 6 weeks.
Glean® (chlorsulfuron)	28-42	High. Persists longer in high pH soils. Weed control longer than Logran.
Diuron	90 (range 1 month to 1 year, depending on rate)	High. Weed control will drop off within 6 weeks, depending on rate. Has had observed longlasting activity on grass weeds such as black/stink grass (Eragrostis spp.) and to a lesser extent broadleaf weeds such as fleabane.
Atrazine	60–100, up to 1 year if dry	High. Has had observed long lasting (>3 months) activity on broadleaf weeds such as fleabane.
Simazine	60 (range 28–149)	Med./high. 1 year residual in high pH soils. Has had observed long lasting (>3 months) activity on broadleaf weeds such as fleabane.
Terbyne® (terbulthylazine)	6.5–139	High. Has had observed long lasting (>6 months) activity on broadleaf weeds such as fleabane and sow thistle
Triflur® X (trifluralin)	57–126	High. 6–8 months residual. Higher rates longer. Has had observed long lasting activity on grass weeds such as black/stink grass (Eragrostis spp.).
Stomp® (pendimethalin)	40	Medium. 3–4 months residual.
Avadex® Xtra (triallate)	56–77	Medium. 3–4 months residual.

42 B Haskins (2012) Using pre-emergent herbicides in conservation farming systems. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0003/431247/Using-pre-emergent-herbicides-in-conservation-farming-systems.pdf

43 B Haskins (2012) Using pre-emergent herbicides in conservation farming systems. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0003/431247/Using-pre-emergent-herbicides-in-conservation-farming-systems.pdf

Herbicide	Half-life (days)	Residual persistence and prolonged weed control
Balance® (isoxaflutole)	1.3 (metabolite 11.5)	High. Reactivates after each rainfall event. Has had observed long lasting (> 6 months) activity on broadleaf weeds such as fleabane and sow thistle.
Boxer Gold® (prosulfocarb)	12–49	Medium. Typically quicker to break down than trifluralin, but tends to reactivate after each rainfall event.
Sakura® (pyroxasulfone)	10–35	High. Typically quicker breakdown than Trifluralin and Boxer Gold, however, weed control persists longer than Boxer Gold.

Table 22: *Minimum re-cropping intervals and guidelines (NOTE: always read labels to confirm).*

Group and type	Product	pH (H2O) or product rate (ml/ha) as applicable	Minimum re-cropping interval (months after application), and conditions
B, sulfonyl urea (SU)	Chlorsulfurons eg Glean®, Seige®, Tackle®	<6.5 6.6–7.5 7.6–8.5	3 months 3 months, minimum 700 mm 18 months, minimum 700 mm
B, sulfonyl urea (SU)	triasulfuron, eg Logran®, Nugrain®	7.6–8.5 >8.6	12 months, >250 mm grain, 300 mm hay 12 months, >250 mm grain, 300 mm hay
B, Sulphonamide	Flumetsulam eg Broadstrike®		0 months
B, sulfonyl urea (SU)	metsulfuron eg Ally®, Associate®	5.6–8.5 >8.5	1.5 months Tolerance of crops grown through to maturity should be determined (small scale) previous season before sowing larger area.
B, sulfonyl urea (SU)	Metsulfuron + thifensulfuron Eg Harmony® M	7.8–8.5 Organic matter >1.7% >8.6 or organic matter <1.7%	3 months Tolerance of crops grown through to maturity should be determined (small scale) previous season before sowing larger area.
B, sulfonyl urea (SU)	Sulfosulfuron eg Monza®	<6.5 6.5–8.5	0 months 10 months

Source: Pulse Australia

Herbicides with long residuals can affect subsequent crops, especially if they are effective at low levels of active ingredient, such as the sulfonylureas. On labels, this will be shown by plant-back periods, which are usually listed under a separate plant-back heading or under the "Protection of crops etc." heading in the "General Instructions" section of the label. [44]

MORE INFORMATION

Avoiding crop damage from residual herbicides

Conditions required for breakdown

Warm, moist soils are required to breakdown most herbicides through the processes of microbial activity. For the soil microbes to be most active they need good moisture and an optimum soil temperature range of 18°C to 30°C. Extreme temperatures above or below this range can adversely affect soil microbial activity and slow herbicide breakdown. Very dry soil also reduces breakdown. To make matters worse, where the soil profile is very dry it requires a lot of rain to maintain topsoil moisture for the microbes to be active for any length of time.

For up-to-date plant-back periods, see Weed control in winter crops.

6.14 Herbicide residues

Pulse growers need to be aware of possible herbicide residues that may affect crop rotation choices or cause crop damage. Herbicide residue impacts are more pressing where rainfall has been minimal. After a dry season, herbicide residues from previous crops could influence choice of crop and rotations more than disease considerations. The opposite occurs after a wet year.

Pulse crop types differ in their sensitivity to residual herbicides, so check each herbicide used against each pulse type. Residues of sulfonylurea herbicides can persist in some soils. These residues can last for several years, especially in more alkaline soils and where there is little summer rainfall. The pulses emerge and grow normally for a few weeks, and then start to show signs of stress.

Picloram (e.g. Tordon® 75-D) residues from spot-spraying can stunt any pulse crop grown in that area. This damage is especially marked in faba beans, where plants are twisted and leaves are shrunken. In more severe cases, bare areas are left in the crop where this herbicide had been used, in some cases more than five years ago. Although this damage is usually over a small area, correct identification of the problem avoids confusion and concern that it may be some other problem such as disease.

In wheat–chickpea rotations, the use of fallow and in-crop residual herbicides such as Broadstrike®, Eclipse®, Flame® Grazon®DS, Lontrel® and metsulfuron (Ally®, Associate®, Lynx®) Harmony®M should be avoided.

The use of long-term residual sulfonylurea herbicides such as Monza®, chlorsulfuron (Glean®, Lusta®), and Logran® in wheat should be avoided when re-cropping to chickpeas. [45]

Stay up to date with chemical labels and recommendations by visiting the APVMA and PubCRIS websites.

6.14.1 Sulfonylurea residues, Group B

Sulfonylurea products include:

- metsulfuron (Ally®, Associate®, Lynx®)
- thifensulfuron plus metsulfuron (Harmony®M)
- sulfosulfuron (Monza®)
- chlorsulfuron (Glean®, Lusta®,
- triasulfuron (Hussar, Atlantis, Logran®)

44 B Haskins (2012) Using pre-emergent herbicides in conservation farming systems. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/agriculture/broadacre-crops/winter-crops/general-information/pre-emergent-herbicides

45 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

Usually, Glean®,or Logran® damage is not serious when these products are used as directed, although there is an increased risk of damage given:

- very dry or drought conditions
- highly alkaline (pH >8.5) soils
- excessive overlapping during application.

Sulfonylurea breakdown occurs by hydrolysis, and is favoured by warm, moist conditions in neutral to acid soils. Residues will tend to persist for longer periods under alkaline and/or dry conditions. Persistence of residues is greater for Glean® and Logran®, than for Ally® or Harmony®M.

Residues are root absorbed and translocated to the growing points; therefore, both roots and shoots are affected.

Moderate residue levels

Plant emergence will be patchy, and the first true leaves elongated and narrow. Plants remain stunted, with severe chlorosis of the uppermost leaves (Photo 12).

Photo 12: *Yellowing of new growth (left) and plant stunting (right).*

Photo: A. Storrie, NSW DPI.

Seedlings develop symptoms as the roots reach the sulfonylurea residue layer in the soil. This may occur in the early seedling stage on heavy clay soils, or slightly later on light sandy soils due to movement of residues down the soil profile. Symptoms are often more severe where there is soil compaction, e.g. in wheel tracks.

Symptoms include:

- spear-tipping of lateral roots (root pruning)
- yellowing of uppermost leaves, which can progress to older, lower leaves in severe cases.
- development of zinc-deficiency symptoms—narrow, cupped leaves
- stunted growth

Highly sensitive crops (in order of susceptibility)

- lentils
- chickpea (0.5 ppb)

Highly susceptible indicator weeds

- brassicas (turnip, mustard, radish)
- red pigweed, mintweed

- native jute
- parthenium weed
- paradoxa grass

Strategy

Avoid using Glean® or Logran® on very high pH soils (pH >8.5) if you intend growing chickpeas after wheat. Reassess risk if Glean® or Logran® has been used and drought conditions have been experienced during the wheat crop and in the subsequent fallow. [46]

6.14.2 Imidazolinone (imi) residues, Group B

Imidazolinone products include:

- imazapic + imazapyr (Midas®, OnDuty®)
- imazamox + imazapyr (Intervix®)
- imazapic (Flame®)
- imazethapyr (Spinnaker®, various imazethapyrs)
- imazamox (Raptor®)

Photo 13: *Spinnaker injury to the emerging new chickpea growth.*

Photo: G. Cumming, Pulse Australia.

Imazethapyr (e.g. Spinnaker®) can be damaging (Photo 13). Damage from residues of other 'imi' products should not be serious when used as directed, although there is an increased risk of damage where:

- plant-back periods or rainfall requirements are not adhered to;
- very dry or drought conditions have prevailed (often 150-200 mm rainfall required);
- soils are highly alkaline (pH >8.5);
- extensive overlapping has occurred during application; or
- heavy rainfall after application concentrates treated soil in plant furrows.

Persistence of imi residues is greater for Intervix® and Midas® or OnDuty® than for Flame®.

46 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

Residues are root-absorbed and translocated to the growing points; therefore, both roots and shoots are affected.

Moderate residue levels

Plant emergence will be patchy, and the first true leaves elongated and narrow. Plants remain stunted, with severe chlorosis of the uppermost leaves.

Low residue levels

Seedlings develop symptoms as the roots hit the imi residue layer in the soil. This may occur in the early seedling stage on heavy clay soils, or slightly later on light sandy soils due to movement of residues down the soil profile. Symptoms are often more severe where there is soil compaction, such as in wheel tracks.

Symptoms include:

- spear-tipping of lateral roots (root pruning)
- yellowing of uppermost leaves, which can progress to older, lower leaves in severe cases
- development of zinc-deficiency symptoms—narrow, cupped leaves
- stunted growth

Highly sensitive crops (in order of susceptibility)

- conventional canola
- lentils
- safflower
- oats

Strategy

Avoid using imi products on very acidic soils if you intend growing chickpeas after a Clearfield® wheat or canola in an area with marginal rainfall. Reassess risk if imi products have been used and drought conditions have been experienced during the prior wheat, canola crop or fallow. Be wary of using imi products in short-term chemical fallows or for summer weed control where chickpeas are to be sown. [47]

6.14.3 Triazine residues (atrazine), Group C

Chickpeas have some tolerance to very low rates of atrazine, but triazine carry-over from previous crops should be avoided (see Photo 14). Atrazine significantly increases the frost sensitivity of the crop. Risk of damage increases where there are low levels of subsoil moisture. Crops in this situation are largely surface-rooted and vulnerable to damage when there is herbicide recharge after each rainfall event.

Photo 14: *Narrowing of the leaflets and multiple branching are signs of triazine residues (left). Similar distortion is seen in the roots (right).*

Photo: G. Cumming, Pulse Australia.

47 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

Atrazine breakdown is strongly influenced by soil type and climatic conditions. Rates of breakdown slow considerably under dry conditions, and can stop altogether under drought.

Atrazine is more persistent under the following conditions:

- alkaline soils (especially pH >8.0)
- increasing clay content (i.e. black earths)
- low soil temperatures
- low soil moisture levels.

Atrazine is root-absorbed and translocated up into the shoots, where it accumulates and inhibits photosynthesis. Plants usually emerge, but begin to show symptoms of stunting and chlorosis at 2–6 weeks of age. Atrazine initially accumulates in the tips and margins of the lower leaves. This results in bleaching and necrosis of the leaf margins. Plants are often stunted and plant growth is slow. Other Group C herbicides such as diuron and fluometuron cause similar symptoms, mainly on the older, lower leaves.

Highly susceptible indicator weeds

- mintweed (turnip, mustard, radish)
- brassicas
- black pigweed. [48]

6.14.4 Group I

Products include:

- 2,4-D products (amines, esters)
- dicamba (e.g. Cadence®)
- triclopyr (e.g. Garlon®)
- fluroxypyr (e.g. Lontrel®)

Residues of 2,4-D persist for a relatively short period, and they can be overlooked. Photo 15 shows residual damage from 2,4-D. There are plant-back periods for various rates of products, but the most important value is the minimal rainfall requirement prior to sowing. In 2006, there was significant 2,4-D damage in chickpeas resulting from an application of a 2,4-D product as a late fallow spray and/or knockdown spray prior to sowing. The damage was due to not having received the minimal rainfall requirement of 15 mm before this period commenced. [49]

48 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

49 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

Photo 15: *Residual 2,4-D damage, showing narrowing and thickening of leaflets on younger growth.*

Photo: J. Flemming, NSW DPI.

6.14.5 Group I residual herbicides

Products include:

- clopyralid (Lontrel®)
- picloram (Tordon® 75-D, Tordon® 242, Grazon® DS)
- aminopyralid + fluroxypy (e.g. Hotshot®)

These products are used for in-crop or fallow weed control and can persist for long periods under dry conditions. Lontrel® is used in canola, wheat, barley, triticale and oats, so care with a subsequent chickpea crop is required. It can persist on crop stubble for long periods and then it can become activated when leached into the soil following rainfall. Lontrel® is being used more often for residual control of fleabane. Picloram residues are relatively stable in the soil, with residues fixed onto clay particles and remaining concentrated in the top 10–15 cm of soil. Residues are slowly broken down by microbial action, with decomposition slowing during the colder, winter months. Up to 25% of the applied dose can persist for up to 12 months, or longer under very dry conditions.

Some symptoms of low-level residue damage are not always readily visible in chickpeas, for example:

- retarded, slow growth
- thickening and callousing of the lower stem, usually just above ground level, which can be accompanied by cracking and splitting of the stem in more severe cases
- proliferation of short, lateral roots.

There may also be some slight twisting and bending of the main stem. Higher rates of residue can also affect leaf shape, with a narrowing and thickening of leaflets. A severe reaction may cause cupping and stunting of leaflets.

Strategy

Avoid using Lontrel® or Grazon® DS in the fallow period prior to chickpeas. [50]

6.14.6 Management of herbicide residues in the soil

Using soil-persistent herbicides can provide very effective weed control; however, issues can arise when sensitive crops are planted in the next season. The main factors that influence whether crop damage occurs are: rainfall from application to sowing, temperature when the soil is wet, soil pH, soil organic matter, the sensitivity of the crop to the herbicide, and the relative persistence of the herbicide in the soil. Risk of damage to subsequent crops is greatest when conditions after application are dry from spring until autumn.

Herbicides can be broken down by chemical and/or microbial means. Both require moisture and temperature to be effective. Herbicides break down more slowly in winter when moisture may be available, but temperatures are low. In a Mediterranean climate, there is usually little or no herbicide breakdown over summer, where temperature is high, but there is no moisture available in the top soil. Most herbicide breakdown will occur in spring and autumn.

To achieve sustained breakdown of herbicides, the top 2 cm of the soil needs to be moist for a period of seven days or more. This is because in summer, the soil microbes shut down due to lack of water and it takes time for their populations to build up again. Small rainfall events in summer will be quickly evaporated from the topsoil, so as a general rule the rainfall events of less than 10 mm in summer should not be counted towards the amount of rainfall required for herbicide breakdown. It is those larger events, typically those of 25 mm or more, which will contribute most to herbicide breakdown.

Soil type and soil pH are also important, as they will affect how far the herbicide moves down the soil profile. Most of the microbial activity occurs in the top few centimetres of the soil and if the herbicide moves below this layer, it may be broken down more slowly. For example, sulfonylurea herbicides are much more mobile in alkaline soils, and this contributes to their longer persistence in alkaline soils. Soil organic matter is important as it provides food for the microbes. Microbial populations are typically smaller in soils with low organic matter than in those with higher organic matter.

Following a dry spring and summer, it is generally those large rainfall events in autumn that do most of the work in breaking down herbicides. The larger these events are and the earlier they occur, the lower the risk of crop damage. One added risk is that the first large rainfall event after a long dry summer will release herbicides into the soil water quickly. Planting too soon after that first large rainfall event can result in greater crop damage than waiting for a week to sow. The re-cropping intervals on product labels are a good guide to the likely risks of crop damage. When in doubt, it is good practice to sow a more tolerant crop. [51]

6.15 Herbicide resistance

Herbicide resistance fact box

- Resistance is the inherited ability of an individual plant to survive and reproduce following a herbicide application that would kill a 'wild type' individual of the same species.

50 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

51 GRDC Update Papers. (2016). Can we beat grass weeds or will they beat us. https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/08/Can-we-beat-grass-weeds-or-will-they-beat-us

- Thirty-six weed species in Australia currently have populations that are resistant to at least one herbicide mode-of-action (MOA).
- As at June 2014, Australian weed populations have developed resistance to 13 distinct MOAs (click here for up to date statistics).
- Herbicide-resistant individuals are present at very low frequencies in weed populations before the herbicide is first applied.
- The frequency of naturally resistant individuals within a population will vary greatly within and between weed species.
- A weed population is defined as resistant when a herbicide at a label rate that once controlled the population is no longer effective (sometimes an arbitrary figure of 20% survival is used for defining resistance in testing).
- The proportion of herbicide resistant individuals will rise (due to selection pressure) in situations where the same herbicide MOA is applied repeatedly and the survivors are not subsequently controlled.
- Herbicide resistance in weed populations is permanent as long as seed remains viable in the soil. Only weed density can be reduced, not the ratio of resistant-to-susceptible individuals. The exception is when the resistance gene(s) carry a fitness penalty so that resistant plants produce less seed than susceptible ones, but this is rare. [52]

Herbicide resistance is the inherited ability of an individual plant to survive a herbicide application that would kill a normal population of the same species. During the 1940s and '50s, Australian agriculture relied heavily on the use of broad-spectrum pesticides to control pests. Selective herbicides began to appear in the mid-1970s and have been a fundamental tool for cropping and pasture since. However, as reliance on chemicals has grown over the years we are continuing to see more weeds that have developed resistance to herbicides. In other words, a number of chemicals that we have available to us have become less useful. Herbicide resistance was first recognised in Australia in 1981 where some annual ryegrass developed resistance to diclofop-methyl (Figure 3). [53]

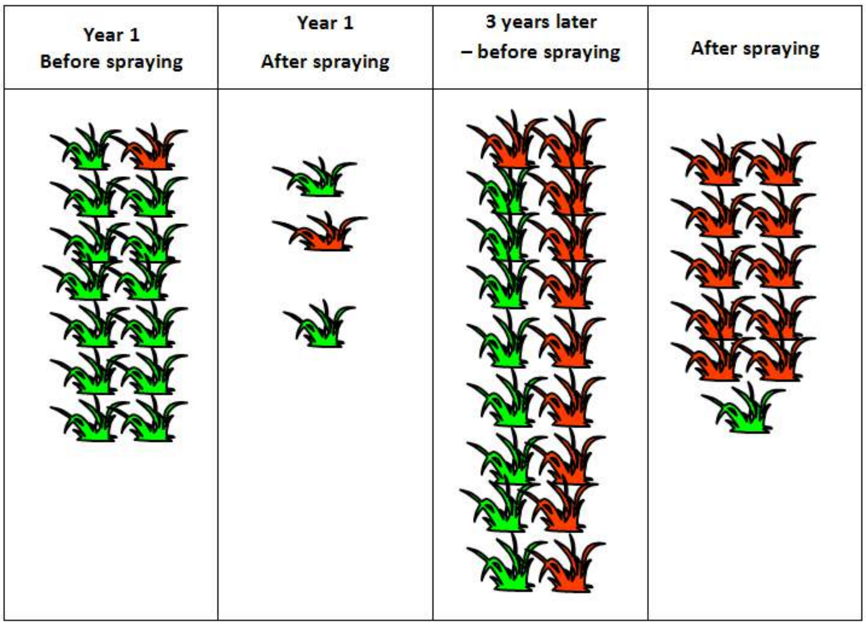

Figure 3: *How a weed population becomes resistant to herbicides.*

Source: GRDC.

52 GRDC. Integrated weed management hub – Section 1: Herbicide resistance. https://grdc.com.au/resources-and-publications/iwmhub

53 Agriculture Victoria. HERBICIDE RESISTANCE AND INTEGRATED WEED MANAGEMENT (IWM) IN CROPS AND PASTURE MONITORING TOOLS. http://agriculture.vic.gov.au/agriculture/farm-management/business-management/ems-in-victorian-agriculture/environmental-monitoring-tools/herbicide-resistance

Herbicide use since the 1980s has seen the development of herbicide resistance across Australia in a range of cropping weeds, including annual ryegrass, wild oats, Indian hedge mustard, wild radish, wild turnip, and prickly lettuce as well as barley grass and capeweed (Table 23). Herbicide resistance is a major threat to Australian grain growers, but whilst herbicide resistance is here to stay, it need not spell the end of profitable cropping. Delaying the onset and/or reducing the impact of herbicide resistant weed populations calls for the implementation of a wide range of weed control strategies, that will in turn help sustain profitable grain production. [54]

Table 23: *Resistance status of a number of weeds. Note: Resistance status will vary from paddock to paddock and not all populations have these characteristics.*

Weed species	Resistance status
Annual Ryegrass (*Lolium rigidum*)	Very high resistance to Group A (e.g. Diclofop) and Group B herbicides (Sulfonylureas). Some resistance to Group D (Trifluralin) and Glyphosate (Group M herbicides).
Wild Oat (*Avena fatua*)	Diclofop-methyl (Group A herbicides) resistance Resistance to Group K (flamprop-methyl)
Barley grass (*Hordeum leporinum*)	Paraquat and Diquat resistance
Capeweed (*Arciotheca calendula*)	Paraquat and Diquat resistance
Barnyard Grass (*Echinochloa crus-galli*)	Resistance to Group C herbicides
Wild Radish	Resistance to chlorosulfron has increased threefold over last four years. Some resistance to Atrazine and 2, 4-D-amine.
Brome grass (*Bromus* spp.)	Resistant to Group A (Verdict) and Group B imi resistance.
Indian hedge mustard, prickly lettuce, wild turnip, sow thistle, black bindweed, silvergrass, summer grass, salvation jane.	New additions of resistant weeds with resistance to one or more groups of herbicides.

Source: AgVic.

Annual ryegrass herbicide resistance

A number of weed species have developed resistance to herbicides. Of the greatest concern is Annual Ryegrass (*Lolium rigidum*) because it has developed cross-resistance to a number of different herbicide groups. Annual Ryegrass is one of the most significant weeds for cropping enterprises—and we are rapidly running out of chemical options to deal with it. Table 26 shows the resistance status of Annual Ryegrass to a number of Group A, B, D, and M herbicides—a herbicide program made up of two to three years use of any of these can fail due to cross resistance. Resistance to trifluralin is increasing rapidly and is high in many areas of South Australia.

54 GRDC (2008). Grain Legume Handbook – Weed control

Table 24: *Estimated number of herbicide applications before resistance develops.*

Product	Low ryegrass number	High ryegrass numbers
Group A	7 to 10	4
Group B		4
Group D (trifluralin)	15	10
Group L		12
Group M		15
Double Knock		30 +

Source: PIR.SA.

Thirty-six weed species in Australia currently have populations that are resistant to at least one herbicide 'mode of action' (MOA) group (Photo 16).

Photo 16: *Pots of annual ryegrass tested for plyphosate resistance; susceptible (left) and strongly resistance (right).*

Photo: Peter Boutsalis, Source: DAFWA.

Herbicide resistance is normally present at very low frequencies in weed populations before the herbicide is first applied. Variation exists within every population, with some individuals having the ability to survive the herbicide application.

A weed population is defined as resistant when a herbicide that once controlled the population is no longer effective (sometimes an arbitrary figure of 20% survival is used). The proportion of herbicide resistant individuals will rise due to selection pressure in situations where one herbicide MOA group is applied repeatedly. [55]

An evaluation of farming systems in low rainfall areas has found that (Table 25):

- As cropping intensity increased, higher average returns are possible, but it is imperative to reduce the number of ryegrass to a very low level prior to, or as soon as possible in the rotation and then using a full range of practices to keep the number low.

55 DAFWA. (2016). Herbicide resistance. https://www.agric.wa.gov.au/grains-research-development/herbicide-resistance?page=0%2C0

- The initial reduction in ryegrass numbers must be carried out with as little selection for resistance as possible. Selective herbicide can be used without the population of ryegrass increasing its resistance if spray topped or green manured before seed set.
- To avoid the build-up in resistance to Group M (glyphosate), the additional cost of the double knock system is justified, particularly in more intensive systems that rely on glyphosate for early weed control in rotations.
- Ryegrass numbers also compete strongly with the crop limiting yield and returns (Photo 17).

Table 25: *Percent Ryegrass Control with different management treatments.*

Treatment	Average % Control Resistant Population	Average % Control
Trifluralin®	30 to 40	80
Trifluralin® + Avadex®	75	85
Boxer Gold®	80 to 90	80 to 90
Sakura®	80 to 90	80 to 90
Double knock (application of glyphosate followed by Sprayseed three days later)	40%	40%
Crop topping		75%
Spray topping (low rate of paraquat or glyphosate) at flowering / milky dough		70%
Brown manure (high rate of glyphosate)		95%
Hay cutting		85%
Stubble burning – grazed		40%
Stubble burning – standing stubble ungrazed		75%
Windrow burning in canola, lupins and beans	85	85% plus
Wheat stubbles from < 2.5 t/ha grain crops		85% plus
Burning chaff dumps		90%
Seed catching		60%
Harrington seed destructor		95%

Source: PIR.SA

Photo 17: *Glyphosate resistant annual ryegrass in crop paddock.*

Source: GRDC.

6.15.1 Herbicide resistant annual ryegrass in the Wimmera and the Mallee

Random resistance surveys have shown that herbicide resistance in annual ryegrass is increasing in Victoria. Randomly selected paddocks in the Wimmera and Mallee were surveyed for resistant weeds in 2015. For annual ryegrass, particular concern is with increasing resistance to trifluralin, Intervix, and glyphosate (Table 26). While trifluralin resistance is becoming widespread in annual ryegrass, we picked up no resistance to any of the other pre-emergent herbicides tested. Glyphosate resistance is now common enough to be picked up in our random weed surveys.

Table 26: *Extent of herbicide resistance in annual ryegrass collected in random surveys in Wimmera and Mallee in 2015. Populations are considered resistant if there is more than 20 per cent survival.*

Herbicides tested	Group	Annual ryegrass populations resistant (%)	
		Wimmera	Mallee
Trifluralin	D	36	23
Boxer Gold®	J + K	0	0
Sakura®	K	0	0
Propyzamide	D	0	0
Hoegrass®	A	80	47
Oust®	B	53	68
Intervix®	B	21	44
Axial®	A	46	10
Select®	A	10	0
Glyphosate	M	9	3

Source: GRDC.

6.15.2 Brome grass resistance rising

Brome grass (Photo 18) is proving to be a headache for grain growers across southern Australia with more plants showing resistance to Group B herbicides. University of Adelaide reports an increasing number of brome grass plants are showing herbicide resistance across the southern and western grain growing districts of Victoria, South Australia, and Western Australia.

Photo 18: *The two main problem species of brome grass in Australia; Diandrus (left) and Rigidous (right).*

Source: GRDC.

Researchers have been collecting brome grass seed samples from fields where farmers were suspicious that herbicide resistance may be the cause of poor herbicide performance.

Samples collected from across the South Australian Mallee region were tested for resistance to current registered herbicides and the tests revealed a high level of herbicide resistance in these weed populations. Herbicide resistance was identified in around 50% of the samples tested.

As well as the South Australian Mallee, brome grass is a significant weed in the Victorian Mallee, Upper Eyre, and in Western Australia. [56]

Integrated management of brome grass

Integrated management of brome grass is much more difficult than integrated management of annual ryegrass. Imidazolinone herbicides, such as Intervix remain the best herbicide options available for brome grass; however, they are Group B herbicides and at high risk of resistance. Trials were conducted, exploring integrated management strategies for brome grass at Balaklava in South Australia over the past three years. The trial consisted of four crop options in rotation with two strategies in each of the crops (Table 27). Clearfield options were used for the cereal phase of the rotation.

56 WeedSmart. 2014. Brome Grass resistance rising – Southern Australia. http://www.weedsmart.org.au/bulletin-board/brome-grass-resistance-rising/

Table 27: *Herbicide strategies investigated for the management of brome in lupins, TT-canola, wheat and barley at Balaklava.*

*Cropping phase	Herbicide strategy (1 & 2)	
	HS1	HS2
Lupins	Simazine pre Haloxyfop post	Simazine pre Haloxyfop post Paraquat crop-top
TT-canola	Atrazine pre Atrazine[ϕa]plus Haloxyfop post	Propyzamide pre Atrazine plus Haloxyfop post Glyphosate crop-top
Wheat	Trifluralin pre Intervix post	Sakura plus Avadex Xtra pre Glyphosate[ϕb] crop-top
Barley	Trifluralin plus metribuzin pre	Trifluralin pre Intervix post

*Wheat and barley cultivars are tolerant to imidazolinone herbicides.

[ϕa]Listed on the label as having suppression of brome grass; [ϕb]Weedmaster DST is the glyphosate product registered for this use.

The two break crops in the rotation were able to reduce the brome grass seed bank, regardless of the strategy used. However, for cereals, the brome grass seed bank was only reduced when Clearfield crops and Intervix herbicide were used (Figure 4). Previous research has demonstrated that you need two consecutive years of good control of brome grass to manage this weed species. Where we had back-to-back break crops (lupins followed by canola) followed by a Clearfield cereal, we were able to reduce brome grass seed production in 2015 to zero. Clearly it is important to use the last remaining Intervix applications on brome grass in the cereal part of the rotation and following a break crop to achieve the best long term result. [57]

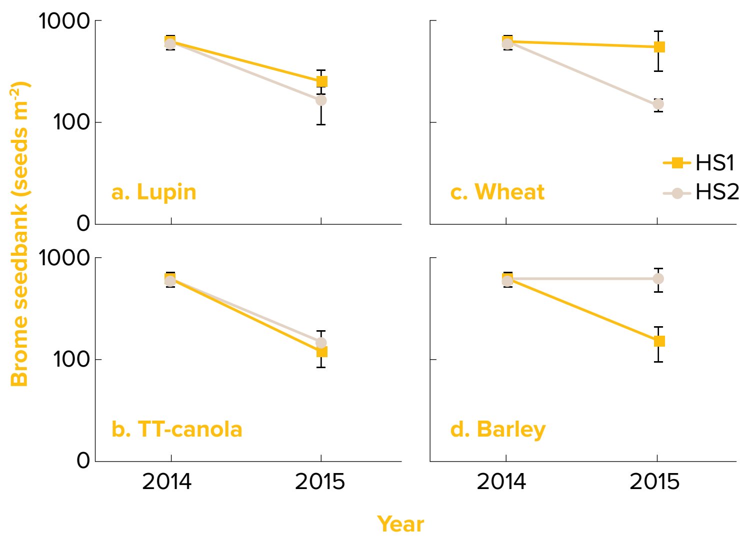

Figure 4: *Change in brome grass seed bank in response to herbicide strategy (HS1 and HS2) in lupin (a), TT-canola (b), wheat (c), and barley (d) crop phases at Balaklava in 2015. Vertical bars represent SE. The initial brome grass seed bank was 626 seeds/m.*

Source: GRDC.

57 GRDC Update Papers. (2016). Can we beat grass weeds or will they beat us. https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/08/Can-we-beat-grass-weeds-or-will-they-beat-us

Glyphosate resistance

Glyphosate resistance was first documented for annual ryegrass (Lolium rigidum) in 1996 in Victoria. Since then, glyphosate resistance has been confirmed in 11 other weed species (Figure 5).

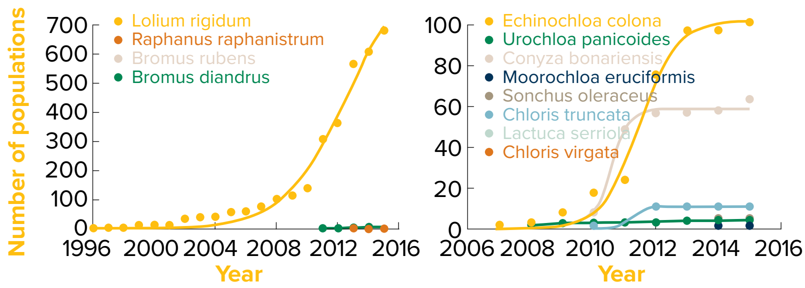

Figure 5: *Increase in confirmed cases of Glyphosate resistance in winter weeds (left) and summer weeds (right) between 1996 and 2016.*

Source: AGSWG.

Resistance is known in eight grass species and four broadleaf species. There are four winter-growing weed species and eight summer-growing weed species. The latter have been selected mainly in chemical fallows and on roadsides.

The most number of resistant populations is for annual ryegrass (Table 28) followed by barnyard grass and then fleabane.

Table 28: *Glyphosate resistant annual ryegrass has occurred in the following situations.*

Situation		Number of Sites	States
Broadacre cropping	Chemical fallow	34	NSW
	Winter grains	393	SA, Vic, NSW, WA
	Summer grains	1	NSW
	Irrigated crops	1	SA
Horticulture	Tree crops	10	SA, NSW
	Vine crops	25	SA, WA
	Vegetables	2	Vic
Other	Driveway	6	SA, Vic, NSW, WA
	Fenceline/crop margin	91	SA, Vic, NSW, WA
	Around buildings	2	NSW
	Irrigation channel/ drain	14	SA, Vic, NSW
	Airstrip	1	SA
	Railway	2	NSW, WA
	Roadside	95	SA, NSW, WA
	Pasture	1	WA

Source: AGSWG.

All of the glyphosate-resistant weed populations have occurred in situations where there has been intensive use of glyphosate, often over 15 years or more, few or no other effective herbicides used and few other weed control practices are used. This suggests the following are the main risk factors for the evolution of glyphosate resistance:

- intensive use of glyphosate—every year or multiple times a year for 15 years or more
- heavy reliance on glyphosate for weed control
- no other weed controls [58]

6.15.3 Practices to minimise herbicide resistance

The threat of herbicide resistance does not mean that herbicides should not be used; however, it does mean farmers should avoid over reliance on herbicides that have the same action on plants ("mode of action"). All herbicide labels now indicate what herbicide group the active ingredient belongs to. Cases of glyphosate resistance in annual ryegrass and of paraquat resistance in barley grass in direct-drill cropping systems sounds a warning on heavy reliance on even "low risk" herbicides.

Growers should aim to use as many different methods of weed control as practical in the overall paddock management including the following:

- rotation of cultivation and herbicide groups
- crop competition
- use of knockdown
- pasture topping herbicides for seedset
- hay making preparation
- grazing
- burning
- seed capture
- crop-topping
- weed wiping (short crops)

Care must be taken when introducing control methods into the overall paddock plan. For example, weed numbers, especially resistant populations, can increase dramatically under pulses due to the poor competition offered by these crops.

Monitoring of weed populations before and after spraying is an important management tool.

Field testing and/or seed testing, as well as planning management strategies, can provide a guide to the resistance status of weed populations. [59]

6.15.4 WeedSmart farming

The Australian grain industry stands at the crossroads with two options. Which direction will it take?

One road is for every grower to make herbicide sustainability their number one priority so that it influences decision-making and practices on all Australian grain farms. Armed with a clear 10-point plan for what to do on-farm, grain growers have the knowledge and specialist support to be WeedSmart.

On this road, growers are capturing and/or destroying weed seeds at harvest. They are rotating crops, chemicals, and modes of action. They are testing for resistance and aiming for 100% weed kill, and monitoring the effectiveness of spray events.

In addition, they are not automatically reaching for glyphosate, they do not cut on-label herbicide rates, and they carefully manage spray drift and residues. Growers are planting clean seed into clean paddocks with clean borders. They use the double-knock technique and crop competitiveness to combat weeds. On this road, the

58 Australian Glyphosate Sustainability Working Group. (2016). Glyphosate resistant weeds in Australia. http://glyphosateresistance.org.au/RegisterSummary.pdf

59 GRDC (2008). Grain Legume Handbook – Weed control

VIDEOS

10. Double knock application – a Grower's Experience.

industry stands a good chance of controlling resistant weed populations, managing difficult-to-control weeds, prolonging the life of important herbicides, protecting the no-till farming system, and maximising yields.

The other option is for growers to think resistance is someone else's problem, or an issue for next year, or something they can approach half-heartedly. If herbicide resistance is ignored, it will not go away. Managing resistance requires an intensive but not impossible effort. Without an Australia-wide effort, herbicide resistance threatens the no-till system, land values, yields, and your hip pocket. It will drive down the productivity levels of Australian farms.

Jump on board WeedSmart and take the road of least resistance.

WeedSmart 10-point plan

1. Act now to stop weed seed set
- Research and plan your WeedSmart strategy.
- Understand the biology of your weeds.
- Be strategic and committed.
2. Capture weed seeds at harvest
- Consider your options—chaff cart, narrow windrow burning, baling, Harrington Seed Destructor.
- Compare the financial cost per hectare.
3. Rotate crops and herbicide modes of action
- Protect the existing herbicide resource.
- Repeated application of effective herbicides with the same MOA is the single greatest risk factor for herbicide resistance evolution.
4. Test for resistance to establish a clear picture of paddock-by-paddock farm status
- Resistance continues to evolve.
- Sample weed seeds prior to harvest for resistance testing.
5. Never cut the rate
- Always use the label rate.
- Weeds resistant to multiple herbicides can result from below the rate sprays.
6. Don't automatically reach for glyphosate
- Consider diversifying
- Consider post-emergent herbicides where suitable.
- Consider strategic tillage.
7. Carefully manage spray events
- Use best management practice in spray application.
- Patch spray area of resistant weeds if appropriate
- No escapes
8. Plant clean seed into clean paddocks with clean borders
- Plant weed-free crop seed
- The density, diversity, and fecundity of weeds is generally greatest along paddock borders and areas such as roadsides, channel banks and fencelines.
9. Use the double knock technique
- Any combination of weed control that involves two sequential strategies
- A second application to control survivors from the first
10. Employ crop competitiveness to combat weeds
- Increase your crop's competitiveness to win the war against weeds.
- Row spacing, seeding rate, and crop orientation can all be tactics to help crops fight. [60]

MORE INFORMATION

WeedSmart Southern Region Guide.

60 WeedSmart 10 point plan. http://www.weedsmart.org.au/10-point-plan/

6.15.5 Testing for herbicide resistance

There are a number of different methods of testing for herbicide resistance. Tests can be performed in-situ (in the paddock during the growing season), on seed collected from the suspect area or by sending live plant samples to a testing service.

Testing can be conducted on-farm or by a commercial resistance testing service.

In-situ testing

An in-situ test can be performed following herbicide failure in a paddock. The test should be done at the earliest opportunity, remembering that the weeds will be larger than when the initial herbicide was applied. Test strips should be applied using herbicide rates appropriate to the current crop growth stage and weed size, plus a double rate. The test strips should only be applied if the weeds are stress free and actively growing. To more accurately assess the level of control, conduct weed plant counts before and after application. Green or dry plant weights can be calculated for more accurate results.

Herbicide resistance seed tests

Seed tests require collection of suspect weed seed from the paddock at the end of the season. This seed is generally submitted to a commercial testing service.

Approximately 3000 seeds of each weed (an A4-sized envelope full of good seed heads) is required for a multiple resistance test. This equates to about one cup of annual ryegrass seed and six cups of wild radish pods.

Syngenta herbicide resistance Quick-Test™

The Syngenta herbicide resistance Quick-Test™ (QT) uses whole plants collected from a paddock rather than seeds, eliminating the problem of seed dormancy and enabling a far more rapid turnaround time. In addition, the tests are conducted during the growing season rather than out of season over the summer. A resistance status result for a weed sample is possible within four to six weeks. The QT, which was developed by Dr Peter Boutsalis while working for Syngenta in Switzerland, is patented in Australia.

For each herbicide to be tested, 50 plants are required. To reduce postage costs, plants can be trimmed to remove excess roots and shoots. Upon arrival at the testing service, plants are carefully trimmed to produce cuttings and transplanted into pots. After appearance of new leaves (normally 5–7 days), plants are treated with herbicide in a spray cabinet. The entire procedure, from paddock sampling to reporting results, takes between 4–6 weeks, depending on postage time and the herbicides being tested. Unlike paddock tests, the QT is performed under controlled conditions, so it is not affected by adverse weather conditions. The age of the plants is also less critical to the testing procedure. Trimming the plants prior to herbicide application means that herbicides are applied to actively growing leaves, thus mimicking chemical application to young seedlings. The Quick-Test™ has been used to test resistance in both grass and broadleaf weed species. During testing, both known sensitive and resistant biotypes are included for comparison.

Quick-Tests can be done with Peter Boutsalis, Plant Science Consulting. [61]

6.16 Grazing for weed control

Grazing is alternative non-chemical option in weed control (Photo 19). Most weeds are susceptible to grazing. Weed control is achieved through reduction in seed-set and competitive ability of the weed. The impact is optimised when the timing of the grazing is early in the life cycle of the weed.

61 DAFWA. (2016). Herbicide resistance. https://www.agric.wa.gov.au/grains-research-development/herbicide-resistance?page=0%2C0

Photo 19: *Sheep grazing pasture.*

Source: GRDC.

Plants vary in their palatability and that under the 'right' stocking rate, animals will selectively graze the more palatable plants. This knowledge is useful when previously grown crops volunteer in the sown crop and herbicides are not available or their use would damage the crop. For example, graze peas in a chickpea crop. The relative palatability for some crops has been determined by the University of Adelaide and are shown in Table 29. The palatability was rated as highly palatable—most of the crop eaten; low palatability—very little of the crop eaten.

For best results:

- introduce sheep early, before crop canopy closes;
- use older sheep;
- use low stocking rates;
- spray weeds along fence line to concentrate sheep in crop;
- remove sheep before they do much damage to crop;
- remove sheep before flowering.

Observe grazing withholding periods if any chemicals are used in crop. [62]

Table 29: *Relative palatability of various crops to sheep.*

Highly palatable	Moderately palatable	Low palatability
Nine weeks after sowing		
Field peas, lathyrus, fenugreek, lentils, canola, wheat, safflower, lupin, blanchefleur, and Languedoc vetch.	Chickpeas	Coriander, faba beans, narbon bean
13 weeks after sowing		
Field peas, lathyrus, canola	Lentils, lupins	Chickpeas, coriander, faba beans, narbon bean, fenugreek

Source: GRDC.

62 GRDC (2008). Grain Legume Handbook – Weed control

6.16.1 Grazing stubbles or failed crops

When putting stock onto crop stubbles or failed crops, there are several considerations, the most important being:

- pulpy kidney
- acidosis, also known as grain poisoning
- nitrates or cyanides in weeds
- wind erosion of soil
- withholding periods

Some simple actions can overcome these issues:

- Ensure that stock have had their 5-in-1 vaccinations and boosters.
- Pulpy kidney is the weakest of the vaccines in 5-in-1, and it is cheap insurance to vaccinate again.
- Ensure that stock have a full rumen prior to going onto a crop.
- This can be easily done by providing hay or stubble as gut-fill.
- This will avoid over gorging on weeds or grain and give the rumen time to adjust to the change in feed.
- Spread large piles of grain out to minimise excessive intakes and risk of acidosis.
- Double-check previous crop chemical treatments and make sure all withholding periods are met before introducing stock.
- Slowly introduce stock to feed by allowing increasing periods over a week, starting with two hours.

Watch stock closely for the first week to ensure no problems occur, including unpalatability, which will result in decreased intake and loss of condition. [63]

63 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

Insect control

Key messages

- Chickpea is more tolerant of most insects than other pulses because of the malic acid they produce. The crop is less susceptible to redlegged earth mite, lucerne flea and aphids than other pulses, though these pests should be monitored and controlled.
- Native budworm (*Helicoverpa punctigera*) is a major pest of pulse crops across southern Australia.
- Chickpea is highly susceptible to native budworm. Crops need to be monitored from flowering through to pod-fill. Small grubs less than 1 cm are damaging. Economic threshold for control can be as low as 1 grub per 10 sweeps of a sweep net.
- When thresholds are exceeded, native budworms can be controlled with insecticides although timing and coverage are both critical to achieving good control.
- The crop will need to be sprayed with an appropriate insecticide if caterpillars are present and pods have started to form.
- Regular monitoring will help determine whether the crop needs to be sprayed. An insecticide application will be necessary if one caterpillar is found in 10 sweeps of the crop. Sweeps should be made while walking through the paddock and consist of a standard sweep of around two metres, sampling the top 15 cm of the crop canopy.
- Synthetic pyrethroids are most effective for native budworm control and will prevent reinfestation for up to six weeks after application. [1]

Chickpea has only one major pest, the native budworm caterpillar, *Helicoverpa punctigera*. Caterpillars do most damage at pod-set through to maturity, and can reduce both grain yield and quality (Table 1).

Insects other than native budworm are rarely a problem in chickpeas post establishment. Chickpeas secrete an organic acid (malic acid) from hairs on their leaves, stems and pods, making the crop unattractive to insects.

Seedlings are most vulnerable to damage:

- before they develop three to four 'true' leaves
- during periods of moisture stress
- when other factors such as low soil temperature or soil compaction limit plant growth. [2]

1 K Regan (2016) Production packages for kabuli chickpea in Western Australia—post planting guide. GRDC, https://www.agric.wa.gov.au/chickpeas/production-packages-kabuli-chickpea-western-australia-post-planting-guide

2 IPM Guidelines (2016) Chickpea—southern region, GRDC, http://ipmguidelinesforgrains.com.au/crops/winter-pulses/chickpea-southern-region/

Table 1: *Chickpea crop stage vulnerability to insect pests. Present: Insect pest present in crop but generally not damaging. Damaging: Crop susceptible to damage and loss caused by insect pest.*

Pest	Crop stage				
	Emergence/ Seedling	**Vegetative**	**Flowering**	**Podding**	**Grainfill**
RLEM	Damaging	Present	Present		
Lucerne flea	Damaging				
Cutworms	Damaging				
Aphids	Damaging	Present	Present		
Thrips	Present	Present	Present		
Native budworm		Present	Present	Damaging	Damaging

(Source: IPM Guidelines)

Insect ID: The Ute Guide

The Insect ID Ute Guide is a comprehensive reference guide for insect pests commonly affecting broadacre crops and growers across Australia, and includes the beneficial insects that may help to control them. Photos have been provided for multiple lifecycle stages, and each insect is described in detail, with information on the crops they attack, how they can be monitored and other pests that they may be confused with. Use of this app should result in better management of pests, increased farm profitability and improved chemical usage. [3]

App features:

- Region selection.
- Predictive search by common and scientific names.
- Compare photos of insects side by side with insects in the app.
- Identify beneficial predators and parasites of insect pests.
- Opt to download content updates in-app to ensure you are aware of the latest pests affecting crops for each region.
- Ensure awareness of international bio-security pests.

Insect ID: The Ute Guide is available on Android and iPhone.

The changing status of pests and the future of pest management in the Australian grains industry

The Australian grains industry is dealing with a shifting complex of invertebrate pests due to evolving management practices and climate change as indicated by an assessment of pest reports over the last 20–30 years. A comparison of pest outbreak reports from the early 1980s to 2006–07 from south-eastern Australia highlights a decrease in the importance of pea weevils and armyworms, while the lucerne flea,

3 GRDC, https://grdc.com.au/Resources/Apps

Balaustium mites, blue oat mites and Bryobia mites have increased in prominence. These changes are the result of several possible drivers. Patterns of pesticide use, farm management responses and changing cropping patterns are likely to have contributed to these shifts. Drier conditions, exacerbated by climate change, have potentially reduced the build-up of migratory species from inland Australia and increased the adoption rate of minimum and no-tillage systems in order to retain soil moisture. The latter has been accompanied by increased pesticide use, accelerating selection pressures for resistance. Other control options will become available once there is an understanding of interactions between pests and beneficial species within a landscape context and a wider choice of 'softer' chemicals. Future climate change will directly and indirectly influence pest distributions and outbreaks as well as the potential effectiveness of endemic natural enemies. Genetically modified crops provide new options for control but also present challenges as new pest species are likely to emerge. [4]

7.1.1 Key Integrated Pest Management (IPM) strategies for chickpeas:

- Tolerate early damage. Chickpeas can compensate for early damage by setting new buds and pods to replace those damaged by pests. Excessive early damage can reduce yields and delay harvest.
- Damage to pods is of more concern than damage to the plant. The grubs chew holes into the soft pod and feed on the developing and filling seed. Yield loss will occur at larval densities lower than those causing a reduction in grain quality (% defective seed). This is because *Helicoverpa* consumes most of a chickpea seed; the remaining damaged seed is generally lost during harvest.
- Monitor larval infestations as mortality of small larvae can be high. Refer to records from successive checks to help interpret check data and make decisions about the need for, and timing of, control.
- Aim for one well-timed spray: Chickpea can tolerate moderate to high numbers of native budworm larvae (10–20 larvae/m^2) through late-vegetative and early-flowering stages. Yield loss is sustained from damage at pod-fill—the most critical stage for protecting the crop.
- Post treatment checks are critical to determine efficacy and possible reinfestation prior to harvest. [5]
- Chickpeas are unique in that they do not host significant numbers of beneficial insects. Small numbers of parasitic flies (tachinids) have been recorded on chickpea, but little else. Therefore, in relation to IPM, there are no in-crop management opportunities via beneficial insects.

7.1 Pest management process

Figure 1 outlines the steps in the pest management process.

4 AA Hoffmann, AR Weeks, MA Nash, GP Mangano, PA Umina. (2008) The changing status of invertebrate pests and the future of pest management in the Australian grains industry. Animal Production Science, 48(12), 1481-1493.

5 IPM Guidelines (2016) Chickpea—southern region, GRDC, http://ipmguidelinesforgrains.com.au/crops/winter-pulses/chickpea-southern-region/

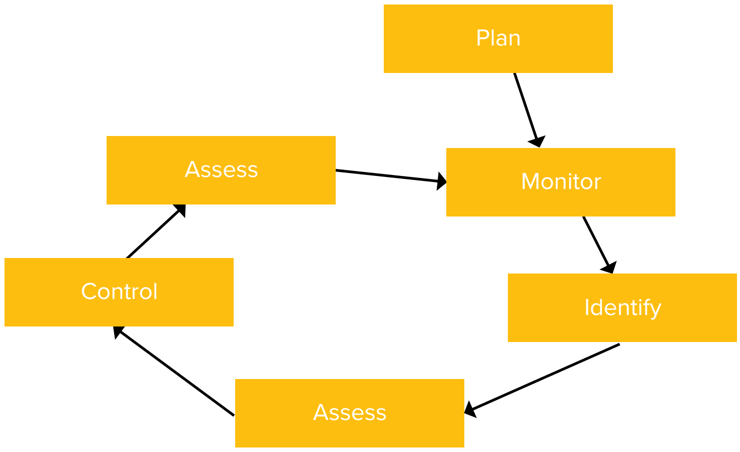

Figure 1: *Pest management process.*

1. Planning
- Be aware of which pests are likely to attack the crop in your region and become familiar with when to monitor for particular pests, what the pests look like, and damage symptoms.
- Assess sampling protocols and plan how you will cope with the logistics of sampling.
- Be aware of the latest management options, pesticide permits and registrations in chickpeas, and any use and withholding-period restrictions.

2. Monitoring
- Scout crops thoroughly and regularly during 'at-risk' periods, using the most appropriate sampling method.
- Record insect counts and other relevant information with a consistent method to allow comparisons over time.

3. Correct identification of insect species
- Identify the various insects present in your crop, whether they are pests or beneficial species, and their growth stages.
- Identify the different larval instars of *Helicoverpa* (very small, small, medium, large).
- Other minor pests of chickpeas should be recorded. These might include locusts, aphids, cutworms, false wireworms, thrips and loopers.

4. Assessing options
- Use the information gathered from monitoring to decide on the control action (if any) required.
- Make spray decisions based on economic threshold information and your experience. Other factors, such as insecticide resistance and area-wide management strategies, may affect spray recommendations.

5. Control
- Ensure that your aerial operators and ground-rig spray equipment are calibrated and set up for best practice guidelines.
- If a control operation is required, ensure that application occurs at the appropriate time of day.
- Record all spray details, including rates, spray volume, pressure, nozzles, meteorological data (relative humidity, temperature, wind speed and direction, inversions and thermals) and time of day.

6. Re-assess and document results

- Assess crops after spraying and record data for future reference.
- Post-spray inspections are important in assessing whether the spray has been effective, i.e. if pest levels have been reduced below the economic threshold. [6]

7.1.1 PestNotes

PestNotes are designed specifically for growers, agronomists and farm advisers. They bring together the best available information and images on more than 50 invertebrate pests of the southern cropping region. These information sheets have been developed through a collaboration between **cesar** and the South Australian Research and Development Institute. [7]

7.2 Legal considerations of pesticide use

Information on the registration status, rates of application and warnings related to withholding periods, occupational health and safety (OH&S), residues and off-target effects should be obtained before making decisions about which pesticide to use. This information is available from the state department chemical standards branches, chemical resellers, the Australian Pesticide and Veterinary Medicine Authority (APVMA) and the pesticide manufacturer.

This section provides background to some of the legal issues surrounding insecticide usage, but it is not exhaustive. Specific questions should be followed up with the appropriate staff from your local state department.

7.2.1 Registration

All pesticides go through a process called registration, where they are formally authorised (registered) by APVMA for use:

- against specific pests
- at specific rates of product
- in prescribed crops and situations
- where risk assessments have evaluated that these uses are:
- effective (against the pest, at that rate, in that crop or situation)
- safe, in terms of residues not exceeding the prescribed maximum residue level (MRL)
- not a trade risk.

For more information see www.apvma.gov.au.

MORE INFORMATION

www.apvma.gov.au

7.2.2 Labels

A major outcome of the registration process is the approved product label, a legal document that prescribes the pest and crop situation in which a product can be legally used, and how.

MSDS

Material Safety Data Sheets (MSDS) are also essential reading. These document the hazards posed by the product, and the necessary and legally enforceable handling and storage safety protocols.

Permits

In some cases a product may not be fully registered but is available under a permit with conditions attached, which often requires the generation of further data for eventual registration.

6 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

7 PestNotes, http://www.cesaraustralia.com/sustainable-agriculture/pestnotes/

Always read the label

Apart from questions about the legality of such an action, the use of products for purposes or in manners not on the label involves potential risks. These risks include reduced efficacy, exceeded MRLs and litigation. Pesticide-use guidelines are on the label to protect product quality and Australian trade by keeping pesticide residues below specified MRLs. Residue limits in any crop are at risk of being exceeded or breached where pesticides:

- are applied at rates higher than the maximum specified;
- are applied more frequently than the maximum number of times specified per crop;
- are applied within the specified withholding period (i.e. within the shortest time before harvest that a product can be applied); or
- are not registered for the crop in question. [8]

7.3 Aphids

Aphids are small insect pests with oval-shaped green, brown or black bodies. Often occurring in colonies, aphids suck on sap, causing loss of vigour, and in some cases yellowing, stunting or distortion of plant parts (Table 2). Honeydew (unused sap) secreted by the insects can cause sooty mould to develop on leaves. When aphids transmit viruses, the impact on crop growth and yield can be significant. The earlier the transmission of virus, the greater the potential impact.

Direct aphid feeding rarely causes major damage to broadacre crops, and control measures are generally unnecessary, as parasitoids and predators keep populations in check. Exceptions occur when aphid populations are extreme (particularly early) or the compensatory ability of the crop is compromised by stress (particularly moisture stress), and aphid impact on flowering or pod-set/fill may be significant.

Pulse aphids:

- Cowpea aphid *Aphis craccivora*
- Blue green aphid *Acyrthosiphon kondoi*
- Green peach aphid *Myzus persica*
- Pea aphid *Acyrthosiphon pisum*

Table 2: *Susceptibility of pulse crops to aphids according to growth stage.*

Pre-Plant/ Plant	Seedling	Vegetative	Budding/ Flowering
Aphids can transmit viruses.	Cowpea aphid: Colonies start in growing points. Blue green aphid: infest growing tips first then move down stems to the crown as numbers build up. Risk of large infestations is higher if weather conditions are mild and hosts abundant.	In lupins direct feeding during flowering can cause flower abortion and poor pod-set. Heavily infested crops may show signs of wilting—more severe in water-stressed crops. Early colonisation by virus-infected aphids may result in yield losses from virus infections; bean yellow mosaic virus infection (BYMV) or cucumber mosaic virus (CMV). Look for aphids on stems and lower leaves.	Most sensitive crop stage to damage: reduce flowering reduce or prevent pod-set and pod-fill Look for aphids on stems, lower leaves, buds and flowering heads.

(Source: IPM Guidelines)

8 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

Cowpea aphids are black, or dark grey-green, sometimes with a white 'dust' over them (Figure 2). Dense colonies can develop on individual plants, or in well-defined patches. Infestations start in the growing tip, and spread down the stem, causing leaf bunching and stem twisting. Cowpea aphids tolerate warm dry weather, and can be severe on water-stressed plants. Water stress and warm weather before flowering can result in heavy, extensive infestations.

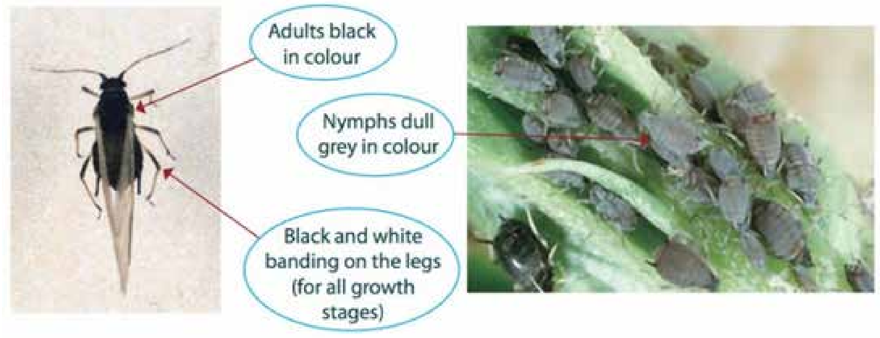

Figure 2: *Distinguishing characteristics/description of cowpea aphids*

Source: Bellati *et al.* 2012 in cesar

Green peach aphid (GPA), *Myzus persicae,* is a widely recognised pest of grains and horticultural crops. It came into particular prominence ss the vector of Beet western yellows virus, which severely impacted many canola crops across south-eastern Australia.

GPAs are waxy green (except the winged adults, which are almost black) (Figure 3). Occasionally, colours of individual wingless GPAs can range from a pale yellow-green to an orange-red. They usually feed in buds and flowers, and do not often form large dense colonies. Generally they are widespread, in low numbers, rather than well-defined patches. They tolerate cool/moist and warm/dry conditions.

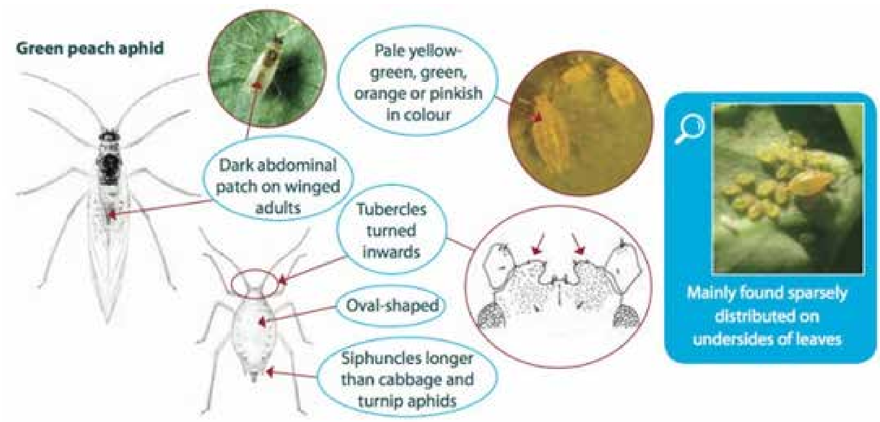

Figure 3: *Distinguishing characteristics/description of GPA*

Source: Bellati *et al.* 2012 in cesar

GPA is resistant to many groups of pesticides. DNA resistance testing is ongoing; to date we have tested 45 populations. Results continue to indicate resistance to the three chemical groups tested: synthetic pyrethroids (e.g. alpha-cypermethrin), organophosphates (e.g. dimethoate) and carbamates (e.g. pirimicarb and imidacloprid). You can view updated resistance maps for SA, Victoria and NSW by clicking here. Despite the high resistance levels to organophosphates across all regions, in some cases, growers have successfully used high levels of dimethoate (i.e. 800 ml/ha) to control GPA populations. Conversely, a similar rate of dimethoate has been reported to offer little control against a GPA population, again in an area where organophosphate resistance was identified. This further serves to illustrate the complexity of this resistance mechanism, and that test strip spraying and working on a case-by case scenario is currently the most effective way to manage resistance in this chemical group. [9]

Life cycle

Winged aphids fly into crops from surrounding vegetation and pastures. Spring population size depends on autumn and winter conditions. Long autumn growing periods allow early build-up and spread of aphids. Mild (not cold) winters allow further development and spread of winged aphids, which can establish many small colonies of wingless aphids throughout a crop. Reproduction is rapid if plant growth and spring weather is favourable, until the colonies are large, and winged aphids redevelop. All aphids are female and give birth to live young, without mating. Viruses carried by flying aphids are transmitted to plants as they feed and establish colonies. Wingless aphids feeding on infected plants can also crawl to healthy plants (through the canopy or after falling to the ground) and spread disease. Viruses can be brought into crops from outside paddocks, or spread within a crop from infected plants.

Damage

Aphids feeding on crops can cause yield loss before plant symptoms become obvious. Large colonies, with more than 40 aphids per stem, cause distortion of leaves, stems and flowers. By the time such symptoms are evident, there will have been yield loss that cannot be recovered by spraying to control the aphids. The crop should be treated before aphid numbers increase markedly. The economic loss over a paddock depends on the area infested, and on the numbers of aphids in each growing tip or bud. Yield losses are greater if virus transmission also occurs. Virus infection causes additional plant symptoms. Aphid feeding slows growth, distorts flowers, and reduces pod-set and fill. Viruses transmitted by the aphids cause a range of symptoms, including 'shepherds crook', stunting, and leaf yellowing. Low numbers of aphids can spread viruses, whereas larger widespread populations are needed to cause direct feeding loss. Virus diseases can cause significant yield loss in pulses. Aphids can carry and spread these diseases, at population levels that cause little damage from direct feeding.

Widespread infestations of GPA during autumn and winter of 2014 contributed to an outbreak of Beet western yellows virus (BWYV, syn. Turnip yellows virus) in southern Australia. Canola crops across the lower and mid-north regions of South Australia, the Eyre Peninsula, western Victoria and some parts of NSW have been severely affected by the virus.

The severity of the current BWYV outbreak is possibly due to a combination of the following factors:

- summer rainfall which resulted in a 'green bridge' of weed hosts of BWYV and aphids;
- the early start to the season and early sowing;
- very mild autumn conditions which contributed to early (and extended) levels of aphid activity through till late June;
- crop management practices;
- the prevalence of insecticide resistance in GPA (particularly to pyrethroids, organophosphates, carbamates); and
- the low proportion of canola seed treated with imidacloprid (Gaucho®) in some areas.

In South Australia, canola crops in the lower and mid-north were the most severely impacted, followed by the South Australian/Victorian Mallee, and some yield loss from BWYV infection has also occurred in canola crops on Eyre Peninsula.

BWYV is an aphid-borne virus that causes yield and quality losses in canola crops. It also infects other crop and pasture species including mustard, chickpea, faba bean,

9 cesar (2014) Green peach aphid. PestFacts south-eastern. Issue 11 08 October 2014, http://cesaraustralia.com/sustainable-agriculture/pestfacts-south-eastern/past-issues/2014/pestfacts-issue-no-11-8th-october-2014/green-peach-aphid/

VIDEOS

1. Green peach aphids and beet western yellows virus.

field pea, lentil, lupin, lucerne, medic and subterranean clover. BWYV is found widespread throughout southern and eastern Australia.

BWYV infection causes reddening, purpling or yellowing of lower leaves of canola plants. Plants infected early (well before flowering) are often pale and stunted and these plants produce few flowers or seeds. Symptoms are milder and stunting is lacking with late infection. In late infections there is minimum effect on yield. Leaf symptom type and severity differ depending on plant age at infection, environmental conditions and the canola variety involved. Symptoms of BWYV in canola can be confused with those caused by nutrient deficiencies, waterlogging or other plant stresses that cause yellowing, reddening or purpling of lower leaves.

BWYV is not seed-borne and survives from one growing season to the next in over-summering canola, broad-leafed weed species, and perennial legume pastures. BWYV is spread by several aphid species that colonise canola including GPA which is the principle vector. The virus infects the phloem cells in a plants' vascular system but not its other tissues. BWYV is transmitted persistently, i.e. once an aphid feeds on the phloem cells of an infected plant, it acquires the virus. When the infective aphid probes the phloem of a healthy plant, it infects the plant and continues transmitting BWYV for the rest of its life. [10]

Control

Aphid numbers can rise and fall rapidly, mainly in response to weather conditions, so they are virtually impossible to predict beyond a few days. The potential for grain yield loss is high if five or more aphids are found in 30% of buds on the main stem or first branches of a plant, and 15% or more of the crop is affected at this level. Waiting until colonies are large, and plant damage symptoms are obvious, is too late; yield loss has occurred and cannot be recovered. The type of aphid does not matter. GPA is more difficult to kill than the other aphids, and higher rates of aphicide should be used. An aphicide that does not kill beneficial insects (wasps, ladybeetles, lacewings, hoverflies) is preferred. It may be necessary to apply aphicide twice in a season, as each set of buds and flowers develops. Spot spraying may be effective. CMV is carried over in seed from infected plants, and can also be transmitted by aphids. Within a crop, aphids spread CMV from plants growing from infected seed. Usually the disease is localised and patchy, but yield losses due to the virus can exceed 50% if the infection spreads throughout a paddock early on. Recommended management strategies include early seeding, high seeding rates to generate dense stands, use of uninfected seed, and strategic application of aphicides to kill aphids in late winter/early spring. BYMV spreads into paddocks from neighbouring pastures. It is usually restricted to paddock edges, but occasionally widespread infections occur, resulting in severe yield loss. Sparse crops (low seeding rate, seedling loss) are especially vulnerable. Management strategies include high seeding rates to generate dense stands, cereal barriers around the crop, heavy grazing of adjoining pasture paddocks to reduce aphid numbers, and strategic aphicide sprays. [11]

7.3.1 Bluegreen aphid

Bluegreen aphids (BGA) are relatively large (up to 3 mm), matt blue-green, with a pair of slender tubes like exhaust pipes (cornicles) projecting from the back to beyond the tip of the abdomen (Figure 4). Winged aphids fly into pastures or crops and start colonies of wingless aphids, which cause damage. Overcrowding or plant deterioration triggers the development of new winged aphids which migrate to establish new colonies. Winged aphids can spread viruses.

10 B Coutts, R Jones, P Umina, J Davidson, G Baker, M Aftab (2015) Beet western yellows virus (synonym: Turnip yellows virus) and green peach aphid in canola. GRDC Update Papers 10 February 2015, https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2015/02/Beet-western-yellows-virus-synonym-Turnip-yellows-virus-and-green-peach-aphid-in-canola

11 PestWeb Lupin aphids. DAFWA, http://agspsrv34.agric.wa.gov.au/ento/pestweb/Query1_1.idc?ID=1923328529

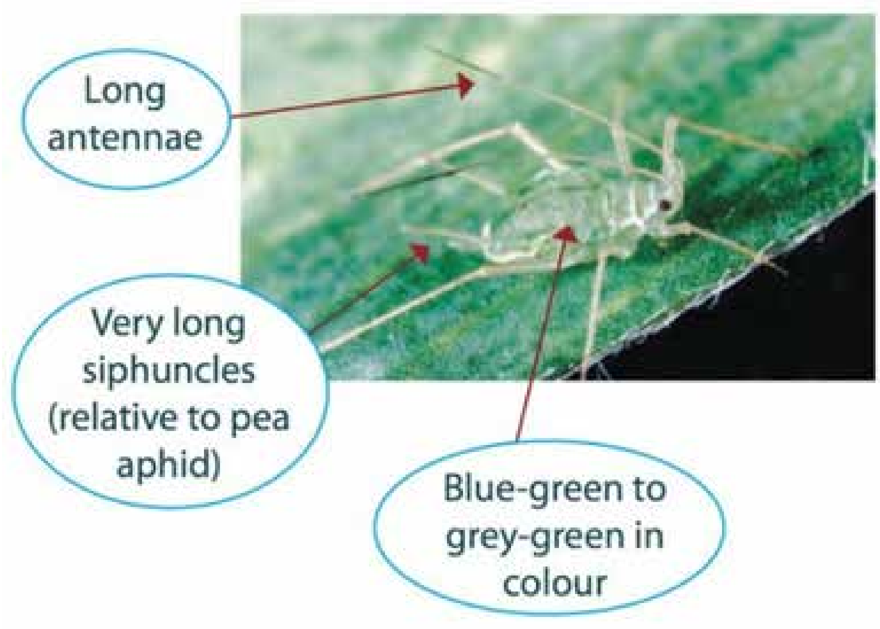

Figure 4: *Distinguishing characteristics/description of bluegreen aphid*

Source: Bellati *et al.* 2012 in cesar.

Life cycle

Like other aphids, all BGA are females and give birth to live young, without having to mate. Reproduction rates are very high so numbers increase rapidly when conditions are favourable. BGA survive hot dry summers in low numbers on sheltered host plants, usually as winged aphids. Migration into germinating annual legumes or lucerne occurs in autumn, and large colonies can develop if it is warm and mild. Winter cold slows reproduction until spring, when populations grow rapidly on favourable plants. During heavy infestations, plants can be covered with white speckles, which are cast-off aphid skins. The number of winged aphids flying between paddocks also increases throughout spring; these can be caught with 'sticky' traps.

Damage

Annual medics, lucerne, subterranean clover and lupins are susceptible to BGA. In lucerne and medics, heavy infestations cause stunted growth, leaf curling and leaf drop. Dry matter production can be reduced. In subterranean clover, leaves wilt before turning grey-brown and dying, becoming dry and 'crisp'. Pastures take on a patchy burnt appearance. Seed yield of annual species can be reduced by 20–80%. The higher the legume content and the lighter the grazing pressure from flowering onwards, the greater the risk of aphid damage. Ungrazed swards with more than 50% legume dominance are at greatest risk in spring. BGA favour growing tips of medic or lucerne, while in subterranean clover they are widely dispersed under the canopy, particularly on flower/burr peduncles.

Control

For lucerne, sow resistant or tolerant cultivars. Parasitic wasps (Photo 1), ladybeetles, lacewings, hoverflies and fungus diseases exert useful biological control. Aphid resistant annual medics and subterranean clover are not common, so insecticides may be needed in lightly or ungrazed spring pastures, if maximum seedset and spring dry matter production is wanted. Redlegged earth mite can cause similar spring losses, and may also be present. If BGA is the predominant pest, use insecticides that do not kill aphid parasites and predators; for mixed infestations, systemic chemicals that control aphids and mites should be used.

Photo 1: *Bluegreen aphid and parasitoid wasp*

Source: cesar

7.3.2 Management

Monitoring

Monitor terminals/growing tips.

- Check at least 5 points in the field and sample 20 plants at each point.
- Check regularly at different locations across a field as populations are often patchy.
- Aphids are often first observed along the edges of crops. Inspect crops from crop edge to centre of paddock. Infestations may be patchy or in 'hot spots'.
- Infestations can be reduced by heavy rain. If rain occurs when spray decision is made but not carried out monitor again to determine is spray still required.
- Record the number of large and small aphids (adults and juveniles), the number of beneficials as well as parasitised aphids (mummies), and the impact of infestation on crop.

Repeat sampling provides information on whether the population is increasing (lots of juveniles), stable or declining (lots of adults and winged adults). [12]

Table 3 summarises control strategies against aphids.

12 IPM Guidelines (2016) Aphids in pulses. GRDC, http://ipmguidelinesforgrains.com.au/pests/aphids/aphids-in-pulses/

Table 3: *Best Bet IPM strategies for controlling aphids in winter pulses in the southern growing region.*

	Post Harvest, pre-sowing	Establishment – vegetative	Flowering - maturity
Aphid vectors and virus source	Control green bridge (in fallows) Sow virus – free seed Sowing into standing stubble may reduce aphid landing	Asses risk of aphid outbreak High risk when: Warm, mild conditions Abundant weed hosts Nearby food sources eg. Clover/medic Aim to close canopy and minimise gaps to outcompete infected plants	Conserve and monitor beneficials that suppress aphids. Use of broad spectrum pesticides may flare aphids. Check post-application for signs of flaring
Aphids – direct damage (not virus) Cowpea Green peach Blue-green Pea aphid	Remove green bridge (aphid hosts) to minimise build up during autumn and spring. Sowing into standing stubble may reduce aphid landing and delay aphid build up in crops.	Control in-crop weeds to minimse sources of aphids Beneficials suppress low population and reduce the chance of outbreaks. High nitrogen may make the crop more attractive to aphids	Monitoring: Conserve and monitor beneficials that suppress aphids. If not control is required, use soft options (eg. Pirimicarb). Use of broad spectrum pesticides may flare aphids. Check post-application for signs of flaring. Note: knowledge of damaging levels is limited.

Source: IPM Guidelines

Chemical control:

- Systemic insecticides are the preferred chemical control (aphids often shelter in spray-inaccessible areas of the plant). However in very dry conditions translocation of chemicals may be impaired and insecticide will be less effective.
- If chemical control is required, consider aphid specific products (e.g. pirimicarb) to preserve beneficials. Refer to the beneficial impact table.
- If heavy rain and cool temperature are forecast consider delaying spray decisions until after rain and monitor again.
- Seed treatments and border spraying (autumn/early winter) when aphids begin to colonise crop edges may provide sufficient control.
- Controlling aphids to prevent virus is not an economic proposition as a small number of aphids can transmit virus and these populations could establish without being detected.
- Rotate chemicals to prevent resistance. GPA potentially has resistance to SPs and OPs. [13]

13 IPM Guidelines (2016) Aphids in pulses. GRDC, http://ipmguidelinesforgrains.com.au/pests/aphids/aphids-in-pulses/

Cultural control:

- Control alternative hosts (wet autumn and spring promotes the growth of weed hosts—when weed hosts dry off aphids move into crops). Legume pasture species are also hosts.
- Sow crops early where possible to enable plants to flower before aphid numbers peak.
- Sow clean seed tested for CMV in lupins or Pea seed-borne mosaic virus (PSbMV) in field peas.
- Cover of bare ground through rapid canopy development assists in deterring aphids, e.g. narrow rows with high seeding rates.
- High intensity rain during crop growth can suppress aphids.
- Research shows that high levels of reflective stubble may deter aphids especially in crops with wide row spacing. [14]

7.3.3 Aphids and virus incidence

Aphids can damage crops by spreading viruses or they can cause direct damage when feeding on plants. Feeding damage generally requires large populations, but virus transmission can occur before aphids are noticed. Pre-emptive management is required to minimise the risk of aphids and their transmission of viruses. Aphids are the principal, but not sole, vectors of viruses in pulses; some viruses are also transmitted in seed.

An integrated approach to aphid and virus management is needed to reduce the risk of yield or quality loss.

Different aphid species transmit different viruses to particular crop types. Viruses are already transmitted before detection, but aphid species identification is important because management strategies can vary. Pulses are annual crops, whereas aphids and the viruses they spread have alternative hosts between seasons. Aphid population development is strongly influenced by local conditions. Early breaks and summer rainfall favour early increases in aphids and volunteers that host viruses, resulting in a higher level of virus risk.

Integrated management practices that aim to control aphid populations early in the season are important in minimising virus spread. Aphids can spread viruses persistently or non-persistently. Once an aphid has picked up a persistently transmitted virus, e.g. BWYV, it carries the virus for life, infecting every plant where it feeds on the phloem. Aphids carrying non-persistently transmitted viruses, e.g. CMV, carry the virus temporarily and only infect new plants in the first one or two probes.

Important vectors for non-persistent viruses in pulse crops include GPA, pea aphid, cowpea aphid and blue green aphid, which will colonise pulse crops (Table 4). Turnip aphid, maize aphid and oat aphid, which are non-colonising species in pulses, may also move through pulse crops, probing as they go, and potentially spreading pulse viruses. GPA and pea aphid are also important in spreading persistently transmitted viruses, depending on the virus involved. [15]

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14 IPM Guidelines (2016) Aphids in pulses. GRDC, http://ipmguidelinesforgrains.com.au/pests/aphids/aphids-in-pulses/

15 GRDC (2010) Aphids and viruses in pulse crops fact sheet. Western and Southern region. GRDC 12 July 2010, https://grdc.com.au/Resources/Factsheets/2010/07/Aphids-and-Viruses-in-Pulse-Crops-Factsheets

Table 4: *Differences in transmission of one persistent and two non-persistent viruses by four aphid species.*

Aphid species	Cucumber mosaic virus (non-persistent)	Pea seed-borne mosaic virus (non-persistent)	Beet western yellows virus (persistent)
Green peach aphid	Yes	Yes	Yes
Pea aphid	Yes	Yes	-
Cowpea aphid	Yes	Yes	Yes
Bluegreen aphid	Yes	-	-

Source: GRDC

7.3.4 Integrated pest management and viruses

An integrated approach with crop, virus and insect management is required to control aphids and viruses in pulse crops.

Minimise the pool of potentially virus-infected plant material near crops by controlling the 'green bridge' of weeds, pastures and volunteer pulses that can harbour viruses and aphids over summer or between crops. This includes weeds around dams, tracks and the margins of crops.

Source clean seed and test retained seed for viruses including CMV, BYMV, Alfalfa mosaic virus (AMV) and PSbMV. Sow tested seed with <0.1% virus infection to reduce the pool of virus-infected material. Field pea seed should have <0.5% PSbMV. Where possible, choose a pulse variety that has virus resistance.

Resistance to CMV seed transmission has been bred into many new lupin varieties, including JENABILLUP⁽ᴾ⁾. YARRUM⁽ᴾ⁾ field pea has resistance to BLRV and PSbMV. Pulse Breeding Australia is increasing its emphasis on developing pulse crop lines with increased virus resistance. Faba bean lines with resistance to BLRV and field pea with resistance to BLRV and PSbMV have been identified and should be commercially available in the future.

Some species of aphids are attracted to areas of bare earth. Use minimal tillage and sow into retained stubble, ideally inter-row to discourage aphid landings. This applies especially to minimising CMV spread in chickpea and lupins.

Seed dressings are probably the best aphid protection strategy compatible with an IPM approach, for example, Gaucho® 350SD insecticide seed dressing on other pulses to prevent aphids attacking emerging seedlings and spreading viruses (e.g. CMV, BLRV and BWYV). However, Gaucho® 350SD is not registered for use in chickpea. Alternatively, a foliar insecticide can be applied early based on forecast reports of the degree of risk. Preferably use a 'soft' insecticide that targets the aphids and leaves beneficial insects unharmed. There is debate over the use of synthetic pyrethroids as a foliar application; they are recommended to prevent BLRV transmission because of so called 'anti-feed' properties that prevent early colonising of crops by pea aphids. However, discouraging colonisation may increase the spread of aphids and, potentially, virus through a crop.

Synthetic pyrethroid insecticides should not be used to control GPA, an important vector of BWYV, as most populations of GPA are resistant. Monitor crops and neighbouring areas regularly. Identify the species of aphid present and their numbers. [16]

16 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

7.4 Blue oat mite

Blue oat mites (BOM) (*Penthaleus* spp.) are species of earth mites, which are major agricultural pests of southern Australia and other parts of the world, attacking various pasture, vegetable and crop plants. BOM were introduced from Europe and first recorded in NSW in 1921. Management of these mites in Australia has been complicated by the recent discovery of three distinct species of BOM, whereas prior research had assumed just a single species.

Blue oat mites are important crop and pasture pests in southern Australia. They are commonly found in Mediterranean climates of Victoria, NSW, SA, WA and eastern Tasmania (Figure 5). There are three main species of BOM: *Penthaleus major*, *Penthaleus falcatus* and *Penthaleus tectus*. These species differ in their distributions.

Figure 5: *The known distribution of blue oat mites in Australia.*

Source: cesar)

Adult BOM are 1 mm in length and approximately 0.7–0.8 mm wide, with 8 red-orange legs. They have a blue-black coloured body with a characteristic red mark on their back (Figure 6). Larvae are approximately 0.3 mm long, are oval in shape and have three pairs of legs. On hatching, BOM are pink-orange in colour, soon becoming brownish and then green.

Figure 6: *Distinguishing characteristics of Blue oat mite.*

Source: Bellati *et al.* (2012in cesar

BOM are often misidentified as redlegged earth mites (RLEM) in the field, which has meant that the damage caused by BOM has been under-represented. Despite having a similar appearance, RLEM and BOM can be readily distinguished from each other. RLEM have a completely black coloured body and tend to feed in larger groups of up to 30 individuals. BOM have the red mark on their back and are usually found singularly or in very small groups.

7.4.1 Damage caused by BOM

Feeding causes silvering or white discoloration of leaves and distortion, or shrivelling in severe infestations. Affected seedlings can die at emergence with high mite populations. Unlike redlegged earth mites, blue oat mites typically feed singularly or in very small groups.

Mites use adapted mouthparts to lacerate the leaf tissue of plants and suck up the discharged sap. Resulting cell and cuticle destruction promotes desiccation, retards photosynthesis and produces the characteristic silvering that is often mistaken as frost damage (Photo 2). BOM are most damaging to newly establishing pastures and emerging crops, greatly reducing seedling survival and retarding development.

Photo 2: *Typical Blue oat mite damage to leaf.*

Source: AgVic

Young mites prefer to feed on the sheath leaves or tender shoots near the soil surface, while adults feed on more mature plant tissues. BOM feeding reduces the productivity of established plants and is directly responsible for reductions in pasture palatability to livestock. Even in established pastures, damage from large infestations may significantly affect productivity.

The impact of mite damage is increased when plants are under stress from adverse conditions such as prolonged dry weather or waterlogged soils. Ideal growing conditions for seedlings enable plants to tolerate higher numbers of mites.

7.4.2 Managing BOM

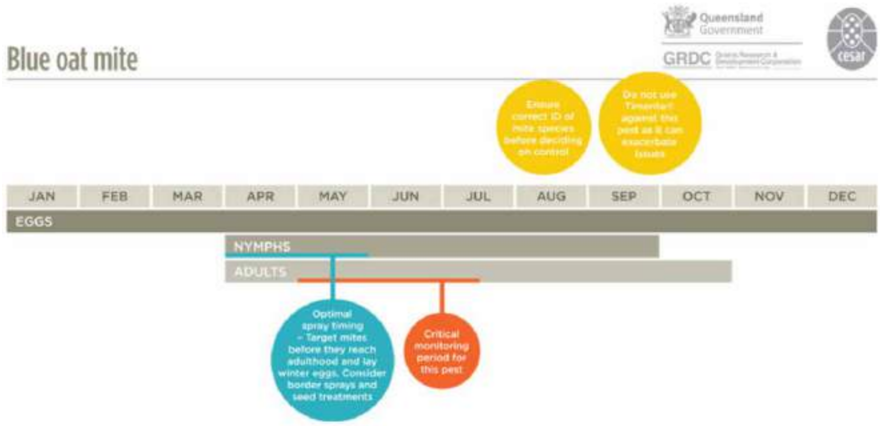


Figure 7: *Critical time periods for managing BOM.*

Source: cesar

Chemical control

Chemicals are the most common method of control against earth mites. Unfortunately, all currently registered pesticides are only effective against the active stages of mites; they do not kill mite eggs.

While a number of chemicals are registered in pastures and crops, differences in tolerance levels between species complicates management of BOM. *P. falcatus* has a high natural tolerance to a range of pesticides registered against earth mites in Australia and is responsible for many control failures involving earth mites. The other BOM species have a lower level of tolerance to pesticides and are generally easier to control with chemicals in the field.

Chemical sprays are commonly applied at the time of infestation, when mites are at high levels and crops already show signs of damage. Control of first generation mites before they can lay eggs is an effective way to avoid a second spray. Hence, pesticides used at or after sowing should be applied within three weeks of the first appearance of mites, as adults will then begin laying eggs. While spraying pesticides in spring can greatly reduce the size of RLEM populations the following autumn, this strategy will generally not be as effective for the control of BOM.

Pesticides with persistent residual effects can be used as bare-earth treatments. These treatments can be applied prior to, or at sowing to kill emerging mites and protect the plants throughout their seedling stage.

Systemic pesticides are often applied as seed dressings to maintain the pesticide at toxic levels within the plants as they grow. This can help minimise damage to plants during the sensitive establishment phase, however, if mite numbers are high, significant damage may still occur before the pesticide has much effect.

To prevent the buildup of resistant populations, spray pesticides only when necessary and rotate pesticides from chemical classes with different modes of action. To avoid developing multiple pesticide resistance, rotate chemical classes across generations rather than within a generation.

Information on the registration status, rates of application and warnings related to withholding periods, OH&S, residues and off-target effects should be obtained before making decisions on which pesticide to use. This information is available from the DEPI Chemical Standards Branch, chemical resellers, APVMA and the pesticide manufacturer. Always consult the label and MSDS before using any pesticide.

Biological & cultural control

Integrated pest management programs can complement current chemical control methods by introducing non-chemical options, such as cultural and biological control.

Although no systematic survey has been conducted, a number of predator species are known to attack earth mites in Australia. The most important predators of BOM appear to be other mites, although small beetles, spiders and ants may also play a role. The French anystis mite is an effective predator but is limited in distribution. Snout mites will also prey upon BOM, particularly in pastures. The fungal pathogen, Neozygites acaracida, is prevalent in BOM populations during wet winters and could be responsible for observed “population crashes”.

Preserving natural enemies when using chemicals is often difficult because the pesticides generally used are broad-spectrum and kill beneficial species as well as the pests. Impact on natural enemies can be reduced by using a pesticide that has the least impact and by minimising the number of applications. Although there are few registered alternatives for BOM control, there are groups such as the chloronicotinyls, which are used in some seed treatments that have low–moderate impacts on many natural enemies.

Cultural controls such as rotating crops or pastures with non-host crops can reduce pest colonisation, reproduction and survival, decreasing the need for chemical control. When *P. major* is the predominant species, canola and lentils are potentially useful rotation crops, while pastures containing predominantly thick-bladed grasses should be carefully monitored and rotated with other crops. In situations where P. falcatus is the most abundant mite species, farmers can consider rotating crops with lentils, while rotations that involve canola may be the most effective means of reducing the impact of *P. tectus*.

Many cultural control methods for BOM can also suppress other mite pests, such as RLEM. Cultivation will significantly decrease the number of over-summering eggs, while hot stubble burns can provide a similar effect. Many broad-leaved weeds provide an alternative food source, particularly for juvenile stages. As such, clean fallowing and the control of weeds within crops and around pasture perimeters, especially of bristly ox tongue and cats ear, can help reduce BOM numbers.

Appropriate grazing management can also reduce mite populations to below damaging thresholds. This may be because shorter pasture results in lower relative

VIDEOS

2. GCTV9: Redlegged earth mites.

humidity, which increases mite mortality and limits food resources. Grazing pastures in spring to less than 2 t/ha Feed On Offer (dry weight), can reduce mite numbers to low levels and provide some level of control the following year. [17]

7.5 Redlegged earth mite (RLEM)

The redlegged earth mite (RLEM), *Halotydeus destructor*, is a major pest of pastures, crops and vegetables in regions of Australia with cool wet winters and hot dry summers. The RLEM was accidentally introduced into Australia from the Cape region of South Africa in the early 1900s. These mites are commonly controlled using pesticides; however, non-chemical options are becoming increasingly important due to evidence of resistance and concern about long-term sustainability. RLEM is a sap-sucking pest of crops and pastures. They often co-exist with blue oat mites. The mites are often gregarious and are found clumped together in large numbers. They disperse quickly when disturbed. [18]

7.5.1 Symptoms

What to look for

Insect adult:

- Adults are 1 mm long with a black body and eight red-orange legs (Photo 3).
- Newly hatched mites are 0.2 mm long with a pinkish-orange body and 6 legs and are not generally visible to the untrained eye.

Photo 3: *Leathery cotyledons with adult RLEM.*

Source: DAFWA 2013

Plant:

- Feeding causes a silver or white discolouration of leaves and distortion. If damage is severe plants shrivel and die (Photo 4).
- Damage is more severe when seedlings are stressed (e.g. cold waterlogged or very dry conditions).

17 Agriculture Victoria (2007) Blue oat mite, http://agriculture.vic.gov.au/agriculture/pests-diseases-and-weeds/pest-insects-and-mites/blue-oat-mite

18 P Umina (2007) Redlegged earth mite. Agriculture Victoria, Ag Note AG0414 January 2007, http://agriculture.vic.gov.au/agriculture/pests-diseases-and-weeds/pest-insects-and-mites/redlegged-earth-mite

Photo 4: *Silver leaf discolouration.*

Source: DAFWA 2013

7.5.2 Damage caused by RLEM

The RLEM is called an earth mite because it spends 90% of its time on the soil surface, rather than on the foliage of plants. The mites feed on the foliage for short periods and then move around before settling at another feeding site. Other mites are attracted to volatile compounds released from the damaged leaves, which results in feeding aggregations. RLEM feeding reduces the productivity of established plants and has been found to be directly responsible for reduction in pasture palatability to livestock. [19]

Large numbers of redlegged earth mite are commonly found in annual pastures at the break of the season and may cause heavy loss of subterranean clover and annual medic seedlings. These species are susceptible throughout the growing season, and can suffer losses in dry matter (10–80%) and seed yield (20–80%) in spring. The greater the legume content of pastures and the lighter the grazing pressure, the higher the risk of loss from mites. They also attack lupins, rape, field peas, serradella (cotyledons only) and vegetables, but normally do not affect grasses or cereals severely. Mites rupture cells on the surface of leaves and feed on exuding sap; affected leaves look silvered, but do not have holes as with lucerne flea attack. Mite damage to seedlings is more severe if plant growth is slowed. This could be caused by cold and/or waterlogging, low seedling density after a false break, low seed banks after a crop, or if pastures or stubble are being reseeded. Capeweed increases their reproductive potential, and legumes in paddocks with a lot of capeweed may be severely damaged, especially where mites can attack smaller clover and medic seedlings from the shelter of large capeweed plants. [20]

In Southern crops RLEM:

- Will damage all field crops and pastures.
- Reduces production and quality of older plants during the growing season.
- Reduces seed yield of legumes in spring.
- Silvering of leaves, distortion of leaf shape in broadleaf crops.
- Affected seedlings can die.

19 P Umina (2007) Redlegged earth mite. Agriculture Victoria, Ag Note AG0414 January 2007, http://agriculture.vic.gov.au/agriculture/pests-diseases-and-weeds/pest-insects-and-mites/redlegged-earth-mite

20 PestWeb Redlegged earth mite. DAFWA, http://agspsrv34.agric.wa.gov.au/Ento/pestweb/Query1_1.idc?ID=247419235

- Seedlings can be killed below ground before they emerge.

7.5.3 Conditions favouring development

Earth mites are active in the cool, wet part of the year, usually between April and November. During this winter-spring period, RLEM may pass through three (sometimes only two) generations, with each generation surviving six to eight weeks (Figure 8).

RLEM eggs hatch in autumn following exposure to cooler temperatures and adequate rainfall. It takes approximately two weeks of exposure to favourable conditions for over-summering eggs to hatch. This releases swarms of mites, which attack delicate crop seedlings and emerging pasture plants.

RLEM eggs laid during the winter-spring period are orange in colour and about 0.1 mm in length. They are laid singly on the underside of leaves, the bases of host plants (particularly stems) and on nearby debris. They are often found in large numbers clustered together. Female RLEM can produce up to 100 winter eggs, which usually hatch in eight to ten days, depending on conditions.

Towards the end of spring, physiological changes in the plant, the hot dry weather and changes in light conditions combine to induce the production of over-summering or 'diapause eggs'. These are stress resistant eggs that are retained in the dead female bodies. Diapause eggs can successfully withstand the heat and desiccation of summer and give rise to the autumn generation the following year. Autumn conditions trigger egg hatching. [21]

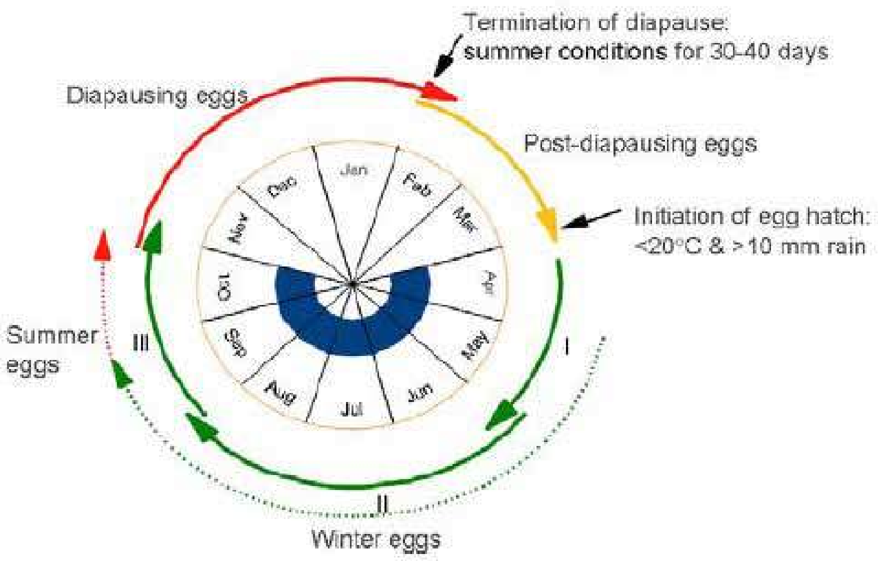


Figure 8: *The life cycle of the redlegged earth mite.*

Source: AgVic

7.5.4 Management of RLEM

Key points:

- Spray only if you need to. RLEM have been detected that have resistance to synthetic pyrethroids. Rotate chemical groups in and between seasons, as this will help to reduce resistance occurring.
- Use insecticide seed treatments for crops and new pastures with moderate pest pressure rather than spraying whole paddocks. This allows for smaller quantities of pesticide to be used that will directly target plant feeding pests.

21 P Umina (2007) Redlegged earth mite. Agriculture Victoria, Ag Note AG0414 January 2007, http://agriculture.vic.gov.au/agriculture/pests-diseases-and-weeds/pest-insects-and-mites/redlegged-earth-mite

- Control weeds before seeding, particularly in late autumn or winter sown crops where RLEM are likely to hatch before seeding. At least one week of bare soil can 'starve out' most of the mite population before crops are sown.
- Control weeds in the crop and along fence lines that provide habitat for mites. A weed free crop will have few mites and over-summering eggs to carry through to the following season.
- Controlled grazing of pasture paddocks that will be cropped next year will reduce mite numbers to levels that are almost as effective as chemical sprays. Sustained grazing of pastures throughout spring to maintain them at levels below 2 tonnes per hectare. Feed on offer (FOO) (dry weight) will restrict mite numbers to low levels.
- Apply insecticides to paddocks that are to be cropped during spring to prevent RLEM populations producing over-summering eggs. This will minimise the pest population for the following autumn. TIMERITE® is a free package that provides a date in spring for a spray application to stop female RLEM from producing over-summering eggs.
- Look at your cropping rotations to decrease reliance on pesticides. The risk is generally highest if paddocks have been in long-term pasture (with high levels of broad-leafed plants) where mite populations have been uncontrolled. Lower risk paddocks that generally do not require mite control are often those which follow a weed free cereal or chickpea crop. [22]

Monitoring

Carefully inspect susceptible pastures and crops from autumn to spring for the presence of mites and evidence of damage. It is especially important to inspect crops regularly in the first three to five weeks after sowing. Mites are best detected feeding on the leaves in the morning or on overcast days. In the warmer part of the day RLEM tend to gather at the base of plants, sheltering in leaf sheaths and under debris. They will crawl into cracks in the ground to avoid heat and cold. When disturbed during feeding they will drop to the ground and seek shelter.

RLEM compete with other pasture pests, such as blue oat mites, for food and resources. Competition within and between mite species has been demonstrated in pastures and on a variety of crop types. This means control strategies that only target RLEM may not entirely remove pest pressure because other pests can fill the gap. This can be particularly evident after chemical applications, which are generally more effective against RLEM than other mite pests.

Chemical control

Chemicals are the most commonly used control option against earth mites. While a number of chemicals are registered for control of active RLEM in pastures and crops, there are no currently registered pesticides that are effective against RLEM eggs.

Autumn sprays:

Controlling first generation mites before they have a chance to lay eggs is the only effective way to avoid the need for a second spray. Hence, pesticides used at or after sowing should be applied within three weeks of the first appearance of mites, before adults begin to lay eggs. Timing of chemical application is critical.

1. Pesticides with persistent residual activity can be used as bare earth treatments, either pre-sowing or at sowing to kill emerging mites. This will protect seedlings which are most vulnerable to damage.
2. Foliage sprays are applied once the crop has emerged and are generally an effective method of control.
3. Systemic pesticides are often applied as seed dressings. Seed dressings act by maintaining the pesticide at toxic levels within the growing plant, which then affects mites as they feed. This strategy aims to minimise damage to plants

22 DAFWA (2015) Diagnosing redlegged earth mite. https://www.agric.wa.gov.au/mycrop/diagnosing-redlegged-earth-mite

during the sensitive establishment phase. However, if mite numbers are high, plants may suffer significant damage before the pesticide has much effect.

Spring sprays:

Research has shown that one accurately timed spring spray of an appropriate chemical can significantly reduce populations of RLEM the following autumn. This approach works by killing mites before they start producing diapause eggs in mid-late spring. The optimum date can be predicted using climatic variables, and tools such as TIMERITE® can help farmers identify the optimum date for spraying. Spring RLEM sprays will generally not be effective against other pest mites.

Repeated successive use of the 'spring spray' technique is not recommended as this could lead to populations evolving resistance to the strategy. To prevent the development of resistance, the selective rotation of products with different Modes of Action is advised.

Biological control

There is evidence of natural RLEM populations showing resistance to some chemicals; therefore, alternative management strategies are needed to complement current control methods.

At least 19 predators and one pathogen are known to attack earth mites in Australia. The most important predators of RLEM appear to be other mites, although small beetles, spiders and ants also play a role in reducing populations. A predatory mite (Anystis wallacei) has been introduced as a means of biological control; however, it has slow dispersal and establishment rates. Although locally successful, the benefits of this mite are yet to be demonstrated.

Preserving natural enemies may prevent RLEM population explosions in established pastures but this is often difficult to achieve. This is mainly because the pesticides generally used to control RLEM are broad-spectrum and kill beneficial species as well as the pests. The chemical impact on predator species can be minimised by choosing a spray that has least impact and by reducing the number of chemical applications. Although there are few registered alternatives for RLEM, there are groups that have low-moderate impacts on many natural enemies such as cyclodienes.

Natural enemies residing in windbreaks and roadside vegetation have been demonstrated to suppress RLEM in adjacent pasture paddocks. When pesticides with residual activity are applied as border sprays to prevent mites moving into a crop or pasture, beneficial insect numbers may be inadvertently reduced, thereby protecting RLEM populations.

Cultural control

Using cultural control methods can decrease the need for chemical control. Rotating crops or pastures with non-host crops can reduce pest colonisation, reproduction and survival. For example, prior to planting a susceptible crop like canola, a paddock may be sown to cereals or lentils to help reduce the risk of RLEM population build-up. Cultivation can also help reduce RLEM populations by significantly decreasing the number of over-summering eggs. Hot stubble burns can provide a similar effect.

Clean fallowing and controlling weeds around crop and pasture perimeters can also act to reduce mite numbers. Control of weeds, especially thistles and capeweed, is important, as they provide breeding sites for RLEM. Where paddocks have a history of damaging, high-density RLEM populations, it is recommended that sowing pastures with a high-clover content be avoided.

Appropriate grazing management can reduce RLEM populations to below damaging thresholds, possibly because shorter pasture results in lower relative humidity, which increases mite mortality and limits food resources.

Other cultural techniques including modification of tillage practices, trap or border crops, and mixed cropping can reduce overall infestation levels to below the economic control threshold, particularly when employed in conjunction with other measures. [23]

Managing resistance

Though RLEM have only been found to show resistance to insecticides in WA, it is important to uphold best practice measure to prevent resistance from emerging closer to home.

Identify your mites

RLEM are often found with other mites, such as blue oat mite (BOM), Bryobia (clover) mite or Balaustium mite, but resistance has only been found in RLEM. In situations where spray failures have occurred, it is important to correctly identify the mite.

Plan ahead to reduce mite numbers

If you prepare paddocks in the preceding season, there will be lower numbers of pests on your crops. Consider the following to reduce pest numbers:

- Control weeds in the crop and along fence lines. Weeds provide habitat for mites. Controlling weeds with herbicides, cultivation or heavy grazing will decrease mite numbers. A weed free crop will have few mites and over-summering eggs to carry through to the following season.
- Controlled grazing of pasture paddocks in the year prior to a cropping year will reduce RLEM numbers to levels similar to chemical sprays. Sustained grazing of pastures throughout spring to maintain Feed on offer levels below two tonnes per hectare (t/ha) (dry weight) will restrict mite numbers to low levels. Control RLEM in spring.
- Applying insecticides to some paddocks during spring to prevent RLEM populations producing over-summering eggs will decrease the pest population in the following autumn. Only specific paddocks should be selected for spring spraying based on FOO levels, future grazing management options, seed production requirements and intended paddock use next season. The routine spraying of all pasture paddocks in spring using TIMERITE® dates to prevent a build-up of mites is unlikely to be sustainable. TIMERITE® is a free package that provides a date in spring for a spraying to stop RLEM from producing over-summering eggs.
- Use cropping rotations to decrease reliance on pesticides. Some paddocks will have a higher or lower risk of RLEM damage depending on previous crop rotations. The risk is generally highest if paddocks have been in long-term pasture (with high levels of broad-leafed plants) where mite populations have been uncontrolled. Lower risk paddocks that generally do not require mite control are often those which follow a cereal or canola weed free crop where conditions are less favourable for mite increase.

What you can do this season

Spray only if you need to

Farmers that currently have populations of resistant RLEM have mostly used repeated applications of SP chemicals as 'insurance' sprays to minimise anticipated pest risks. To decrease the likelihood of resistance developing on your property apply insecticides only on paddocks that have damaging numbers of pests.

Where spraying is needed, rotate chemical groups

For example, rotate between Synthetic Pyrethoids (SP, Group 3A) and Organophosphate (OP, group (Group 1B), e.g. dimethoate or omethoate), in and between seasons, as this will help to reduce resistance build-up. If spraying other pests, such as aphids, try not to use SPs. Consider other chemical options such as pirimicarb.

23 P Umina (2007) Redlegged earth mite. Agriculture Victoria, Ag Note AG0414 January 2007, http://agriculture.vic.gov.au/agriculture/pests-diseases-and-weeds/pest-insects-and-mites/redlegged-earth-mite

Predict hatchings of RLEM on your property to target your control strategy

Knowing approximately when the first autumn hatchings of RLEM is occurring on your property will help to determine if they will coincide with seedling crops. RLEM hatch in autumn from their over-summering egg stage, after adequate rainfall and at least seven days of average temperatures below 20°C. Crops sown in seasons with 'early breaks' with maximum temperatures well above 20°C (for example, canola sown in April) will not be damaged by RLEM.

Use insecticide seed treatments

Use insecticide seed treatments for crops and sown pastures with moderate pest pressure rather than spraying whole paddocks. Seed treatments allow smaller quantities of pesticide to be used that directly target plant feeding pests, allowing any predatory insects to continue their important beneficial role. [24]

7.6 Lucerne flea

The lucerne flea, *Sminthurus viridis* (Collembola: Sminthuridae), is a springtail that is found in both the northern and southern hemispheres but is restricted to areas that have a Mediterranean-type climate. It is thought to have been introduced to Australia from Europe and has since become a significant agricultural pest of crops and pastures across the southern states. It is commonly observed on loam-clay soils, and it is not related to the fleas which attack animals and humans.

The adult lucerne flea is approximately 3 mm long with light green-yellow colouring and an irregular pattern of darker patches over the body (Photo 5). Lucerne fleas are wingless, have globular abdomens and can jump large distances relative to their size. Their mottled colouration, small size and elusive habits can often make detection difficult.

Photo 5: *Yellow-green wingless and globular adults sometimes with dark markings.*

Source: DAFWA 2013

Eggs, which are laid in batches, are covered in a soil layer making them almost impossible to detect in the field. The eggs are yellow-cream and about 0.3 mm in diameter. The newly hatched nymphs are approximately 0.75 mm long and are pale

24 S Micic (2016) Prevent redlegged earth mite resistance. DAFWA, https://www.agric.wa.gov.au/mites-spiders/prevent-redlegged-earth-mite-resistance?page=0%2C2

yellow in colour. Young nymphs resemble adults except they are much smaller in size and will moult several times before reaching maturity.

Lucerne fleas have a characteristic ability to 'spring' off vegetation when disturbed. This is due to a stiff appendage folded under their abdomen called a furcula, which is unfolded with such speed and force that it launches the lucerne flea into the air.

While regional movement of lucerne flea is limited, anecdotal evidence suggests that the incidence of lucerne flea is increasing, even in areas of continuous cropping. The increased adoption of minimum and no-tillage may be contributing to the pest's increased survival and range expansion. [25]

7.6.1 Symptoms

What to look for

Paddock:

- Small jumping bugs that appear early in the season and chew young leaves on heavier textured soils.

Plant:

- Cereals, canola and pasture legumes have chewed leaves with transparent 'windows' (Photo 6).
- Green material completely removed in severe infestations.

Photo 6: *Chewed leaves have transparent 'windows' on a wheat (left) and clover (right) leaf.*

Source: AgVic

Insect adult:

- Adults (3 millimetres) yellow-green, wingless and globular in shape sometimes with dark markings.
  Insects 'spring' off foliage when disturbed. [26]

7.6.2 Damage caused by lucerne flea

Lucerne fleas move up plants from ground level, eating tissue from the underside of foliage. They consume the succulent green cells of leaves through a rasping process, avoiding the more fibrous veins and leaving behind a layer of leaf membrane. This makes the characteristic small, clean holes in leaves which can appear as numerous small 'windows'. In severe infestations this damage can stunt or kill plant seedlings. [27]

- Lucerne flea can kill seedling crops and pastures and re-growth of lucerne.
- Yield loss varies with the growth stage of the plant.

25 G McDonald (1995) Lucerne flea. Agriculture Victoria, Ag Note AG0415 June 1995 Updated June 2008, http://agriculture.vic.gov.au/agriculture/pests-diseases-and-weeds/pest-insects-and-mites/lucerne-flea

26 DAFWA (21015) Diagnosing lucerne flea. https://www.agric.wa.gov.au/mycrop/diagnosing-lucerne-flea

27 G McDonald (1995) Lucerne flea. Agriculture Victoria, Ag Note AG0415 June 1995 Updated June 2008, http://agriculture.vic.gov.au/agriculture/pests-diseases-and-weeds/pest-insects-and-mites/lucerne-flea

- Broadleaf seedlings are most susceptible.
- Young nymphs feed on the soft tissue on the underside of leaves leaving transparent 'windows'.
- Adults and older nymphs chew irregular holes in leaves and can completely defoliate plants. [28]

7.6.3 Thresholds for control

Speculative thresholds: the key is early control because of the impact of seedling vigour on crop performance.

- Establishing pasture: 15 per 100 cm^2 (sampling method not established, but could use that proposed by Taverner *et al.* 1996).
- A suggested threshold for other crops—treat if 50% of leaf area is likely to be damaged. [29]

7.6.4 Conditions favouring development

The lucerne flea has a similar seasonal biology to other important pests of establishing crops, such as the redlegged earth mite. Lucerne fleas generally hatch from over-summering eggs in March–April following soaking autumn rains. They reproduce sexually and, depending on favourable temperatures and moisture availability, may go through as many as three to five generations between autumn and spring. Each generation takes three to five weeks, with each female capable of laying up to three batches of eggs during this time.

Winter eggs are laid in moist crevices on the soil surface in batches of about 20–60, usually under vegetation and debris. Females then excrete a fluid substance containing ingested soil and glandular secretions over the eggs. This acts to both camouflage and protect the eggs.

At the onset of warmer and drier conditions, over-summering eggs are produced which are protected from desiccation by a clay cement layer excreted by females. Consequently, lucerne fleas are more common on heavier loam/clay soils and are rarely found on sandy soils. The protective coating also prevents eggs from hatching when rain is insufficient for lucerne flea development or for the establishment of host plants.

7.6.5 Management of lucerne flea

Monitoring

Monitoring is the key to reducing the impact of lucerne flea. Crops and pastures grown in areas where lucerne flea has previously been a problem should be regularly monitored for damage from autumn through to spring. Susceptible crops and pastures should also be carefully inspected for the presence of lucerne fleas and evidence of damage.

It is important to frequently inspect winter crops, particularly canola and pulses, in the first three to five weeks after sowing. Crops are most susceptible to damage immediately following seedling emergence. Pastures should be monitored at least fortnightly from autumn to spring, with weekly monitoring preferred where there have been problems in previous years.

Lucerne fleas are often concentrated in localised patches or 'hot spots' so it is important to have a good spread of monitoring sites within each paddock. Examine foliage for characteristic lucerne flea damage and check the soil surface where insects may be sheltering.

Some sprays require application at a particular growth stage, so it is also important to note the growth stage of the population. Spraying immature lucerne fleas before

28 IPM Guidelines (2016) Lucerne flea. GRDC, http://ipmguidelinesforgrains.com.au/pests/lucerne-flea-in-winter-seedling-crops/

29 IPM Guidelines (2016) Lucerne flea. GRDC, http://ipmguidelinesforgrains.com.au/pests/lucerne-flea-in-winter-seedling-crops/

they have a chance to reproduce can effectively reduce the size of subsequent generations.

Lucerne fleas compete for food and resources with other agricultural pests such as redlegged earth mites and blue oat mites. This means control strategies that only target one species may not reduce the overall pest pressure because other pests can fill the gap. It is therefore important to assess the complex of pests before deciding on the most appropriate control strategy.

Chemical control

Lucerne fleas are commonly controlled post-emergence, usually after damage is first detected. Control is generally achieved with an organophosphate insecticide (e.g. omethoate). In areas where damage is likely, a border spray may be sufficient to stop invasion from neighbouring pastures or crops. In many cases, spot spraying, rather than blanket spraying, may be all that is required.

If the damage warrants control, treat the infested area with a registered chemical approximately three weeks after lucerne fleas have been observed on a newly emerged crop. This will allow for the further hatch of over-summering eggs but will be before the lucerne fleas reach maturity and begin to lay winter eggs.

In pastures, a follow-up spray may be needed roughly four weeks after the first spray to control subsequent hatches, and to kill new insects before they lay more eggs. Grazing the pasture before spraying will help open up the canopy to ensure adequate spray coverage. The second spray is unlikely to be needed if few lucerne fleas are observed at that time.

Crops are most likely to suffer damage where they follow a weedy crop or a pasture in which lucerne flea has not been controlled. As such, lucerne flea control in the season prior to the sowing of susceptible crops is recommended.

Caution is advised when selecting an insecticide. Several chemicals registered for redlegged earth mites (i.e. synthetic pyrethroids such as cypermethrin) are known to be ineffective against lucerne flea. When both lucerne fleas and redlegged earth mites are present, it is recommended that control strategies consider both pests, and a product registered for both is used at the highest directed rate between the two to ensure effective control.

Information on the registration status, rates of application and warnings related to WHP, OH&S, residues and off-target effects should be obtained before making decisions on which insecticide to use. This information is available from the DPI Chemical Standards Branch, chemical resellers, APVMA and the pesticide manufacturer. Always consult the label and MSDS before using any insecticide.

Biological control

Several predatory mites, various ground beetles and spiders prey upon lucerne fleas. Snout mites (which have orange bodies and legs) are particularly effective predators of this pest (Photo 7). The pasture snout mite (*Bdellodes lapidaria*) and the spiny snout mite (*Neomulgus capillatus*) have been the main focus of biological control efforts against lucerne flea.

Photo 7: *Predatory adult snout mite.*

Photo: A Weeks (cesar), AgVic

There are some examples of this mite successfully reducing lucerne flea numbers. Although rarer, the spiny snout mite can also drastically reduce lucerne flea populations, particularly in autumn.

Three species of predatory mites feed on lucerne flea. They include:

- pasture snout mite (*Bdellodes lapidaria*);
- spiny snout mite (*Neomulgus capillatus*); and
- French anystis mite (*Anystis wallacei*) can provide effective suppression of lucerne flea.

Some field experiments indicate a 70–90% control of lucerne fleas by predatory mites. Other reports suggest that predatory mite activity is rarely effective to reduce lucerne flea impact on seedling crops. Predatory mites are slow to spread and can only do so by crawling. Redistribution of predatory mites is possible using suction machines to collect and transfer mites from established to new sites.

Other beneficials include: ground beetles and spiders. [30]

7.7 Cutworms

Cutworms are plump, smooth caterpillars, of several moth species. They feed on all crop and pasture plants, damaging them near the ground. When mature, they pupate in the soil. Cutworm caterpillars grow to about 40 mm long, but they usually cannot be seen as they hide under the soil or litter by day. Often they can be located by scratching the surface near damaged plants where they can be seen curled up in a defensive position.

Caterpillars with a pink tinge belong to the pink cutworm, Agrotis munda, which has caused widespread damage in agricultural areas north of Perth. The dark grey caterpillars of the Bogong moth, *Agrotis infusa*, have been extremely damaging in most parts of the agricultural areas from time to time. Large numbers of patterned caterpillars belonging to different genera, *Rictonis* and *Omphaletis*, have also been found attacking cereals in agricultural areas.

Adult cutworms are stout-bodied moths with patterned wings. They fly very well and may be seen on window panes at night as they are attracted to lights. [31]

30 IPM Guidelines (2016) Lucerne flea. GRDC, http://ipmguidelinesforgrains.com.au/pests/lucerne-flea-in-winter-seedling-crops/

31 S Micic (2016) Cutworm: pests of crops and pastures. DAFWA, https://www.agric.wa.gov.au/pest-insects/cutworm-pests-crops-and-pastures

Where have they been reported?

In Victoria in 2015, large numbers of cutworms caused extensive damage to a large barley crop north of St. Arnaud in the Wimmera district. The caterpillars chewed seedlings back to ground level, affectively wiping out almost 90% of the paddock. Cutworms were reported attacking a vetch crop near Donald, in the Wimmera. The larvae have completely severed plant stems just above the soil surface, causing significant damage across about 40% of the paddock. Cutworms may also have been responsible for extensive damage to a canola crop near Murtoa, also in the Wimmera district. The crop, which was at the cotyledon stage, experienced significant feeding damage typical of cutworms, with large areas of the paddock requiring re-sowing. Cutworms were also responsible for crop damage around Elmore, in the Northern Country district of Victoria. Several canola paddocks were affected. [32]

7.7.1 Symptoms

What to look for

Insect adult:

- Adult cutworms are stout-bodied moths with patterned wings (Photo 8).

Photo 8: *Cutworm moths.*

Source: DAFWA 2013

Insect larvae:

- Caterpillars are up to 50 mm long, hairless with a dark head.

32 cesar (2014) Cutworms. PestFacts south-eastern. Issue 3 22 May 2014, http://www.cesaraustralia.com/sustainable-agriculture/pestfacts-south-eastern/past-issues/2014/pestfacts-issue-no-3-22nd-may-2014/cutworms/

- They vary in colour and can be:
- dark grey to green body often with lines and/or dark spots running along length
- pale grey-green body with a pinkish tinge, often with lines and/or dark spots running along length
- orange-brown body with diagonal markings (Photo 9).

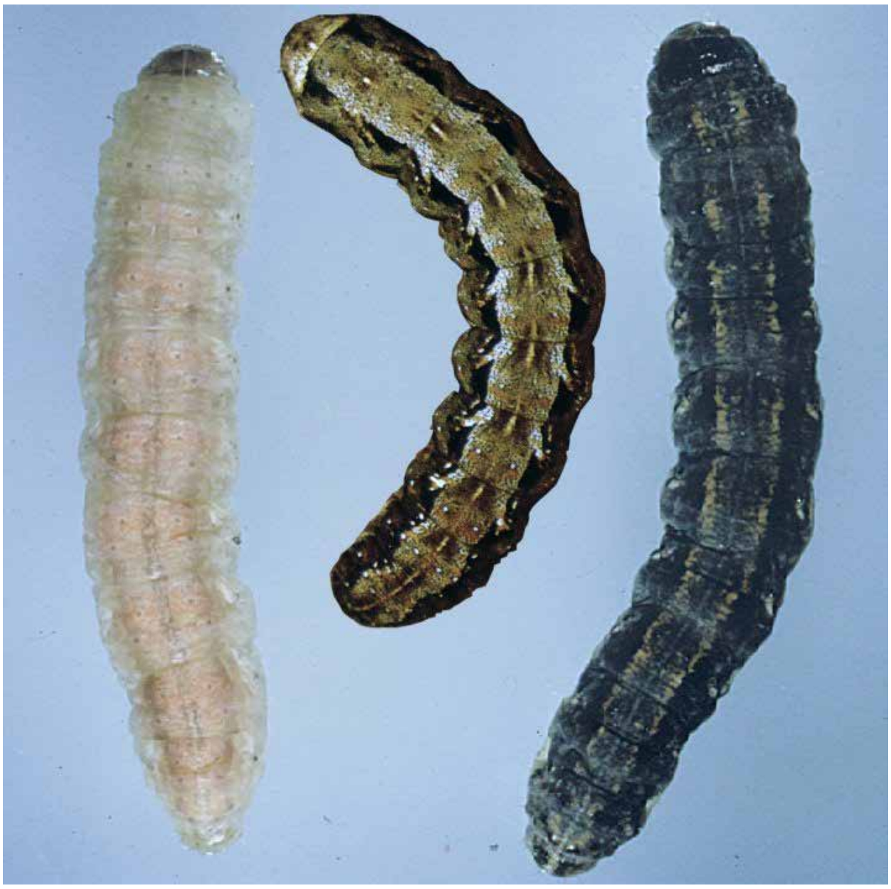


Photo 9: *Larvae of the three main species of cutworm.*

Source: DAFWA 2013

Paddock:

- Commonly patches with plants that have leaves lopped or are cut off at the base (Photo 10).

Photo 10: *Damage often occurs in patches.*

Source: DAFWA 2013

Plant:

- Damage is worst at the seedling stage but can persist for several weeks.
- Larvae hide in the soil during the day, often at the base of lopped plants (Photo 11). [33]

33 DAFWA (2015) Diagnosing cutworm in canola and pulses. https://www.agric.wa.gov.au/mycrop/diagnosing-cutworm-canola-and-pulses

Photo 11: *Lopped lupin and wheat (insert) plants.*

Source: DAFWA 2013

7.7.2 Damaged caused by cutworms:

- Small larvae cause skeletonised or scalloped leaves (damage may be confused with that of lucerne flea or pasture web worm).
- Large larvae sever seedling stems near ground level.
- Large larvae (40–50 mm) are the damaging stage. These larvae may remain below the soil surface feeding on the stem at or below ground level.
- While generally a pest of seedlings (1 to 5 leaf stage), cutworms can occasionally cause damage at tillering and early stem elongation in winter cereals. [34]

Economic and financial considerations

To assist in assessing the economic risk and financial costs associated with various treatment strategies go to MyEconomicTool.

There may be other economic and financial implications that need to be considered when choosing a management option. These may include:

Pre-crop:

- Understand the potential yield losses associated with cutworm feeding damage.
- Assess the costs and benefits of taking preventative action.
- Assess the cost and benefits of controlling summer weeds (green bridge) to reduce potential feed source.

34 IPM Guidelines (2016) Cutworms. GRDC, http://ipmguidelinesforgrains.com.au/pests/soil-insects/cutworms/

In-crop:

- Compare the costs, benefits and risk of each management option against doing nothing or delaying treatment.
- Ignore all previous treatment costs in assessing current management options.
- Undertake a 'what if' scenario analysis to see what impact changing variables, such as grain price and season, have on the net income.

Post-crop:

- Consider using IPM system and include a resistance management strategy into your spray program to reduce the chance of cutworm and other non-target insects becoming resistant. [35]

View these economic considerations in more detail.

7.7.3 Conditions favouring development

Cutworm moths can fly large distances and favour bare or lightly vegetated areas for egg-laying. Moths emerge in late spring or early summer and are often observed entering houses and buildings for shelter over summer. They have one generation per year.

They are most damaging in autumn when large caterpillars (>20 mm) transfer from summer and autumn weeds onto newly emerged crop seedlings.

7.7.4 Thresholds for control

Control is warranted when two larvae per 0.5 metre of crop row are present under visual inspection.

7.7.5 Control

Cutworms are easily controlled by insecticides. They are most damaging in autumn when large caterpillars (>20 mm) transfer from summer and autumn weeds onto newly emerged seedlings.

Monitoring:

- Inspect crops regularly from emergence to establishment. Larvae are active from late afternoon through the night.
- Look for signs of feeding on leaves—if detected, search the soil and stubble in the areas that are damaged, or where the plant stand is thinned.
- Larvae may move into a crop from a neighbouring weedy fallow, particularly as the weeds start to dry off, or are sprayed. Be alert to sources of larvae from outside the field.

Chemical control:

- Treat the crop when seedling loss is nearing minimal plant density crop requirements.
- Treat older plants if more than 50% of plants have 75% or more leaf tissue loss.
- Chemical control is most effective when applied late in the day to maximise likelihood of larvae contacting/ingesting insecticide when they emerge at night to feed.
- Ground-rig applications may provide flexibility to treat just affected areas, or to apply a border spray where larvae are moving into the crop from neighbouring weeds.

Cultural control:

- Control weeds in and around fields prior to planting to reduce potential cutworm infestations.

35 DAFWA (2015) Diagnosing cutworm in canola and pulses. https://www.agric.wa.gov.au/mycrop/diagnosing-cutworm-canola-and-pulses

- Be aware of cutworm movement from sprayed weedy fallow into neighbouring crops.
- Prolonged green feed in autumn allows larvae to develop to a large size by the time crops emerge.
- Aim to control potential hosts at least two weeks prior to planting to ensure larvae do not survive to infest crops.

Biological control

Biological control agents, or beneficials, include fly and wasp parasites, predatory beetles and diseases, continually reduce cutworm numbers but cannot be relied on to give adequate control. Orange and two-toned caterpillar parasites and orchid dupe are key parasitoids. These beneficials may suppress cutworm populations, but are unlikely to prevent crop loss in the event of an outbreak. [36]

7.8 Locusts

Locusts and grasshoppers will cause damage to chickpeas in the same way that they will cause damage to any green material when in plague numbers. Chickpeas may be less vulnerable in the seedling stages than lupins and lentils. However, sheer weight of numbers can lead to significant damage (Photo 12).

Photo 12: *Locust swarms can travel long distances on the wind. Landholders are required to report locust activity.*

Source: Pulse Australia

36 IPM Guidelines (2016) Cutworms. GRDC, http://ipmguidelinesforgrains.com.au/pests/soil-insects/cutworms/

Though locust plagues are infrequent, they can be unpredictable and cause serious damage if not managed.

In spring 2010, Victoria experienced high densities of the pest species Australian plague locust (APL). Swarms across northern Victoria intensified, with swarms reported in the Bordertown-Ouyen, Cullulleraine-Mildura, Sea Lake-Swan Hill and Kerang-Echuca areas (see Figure 9). [37]

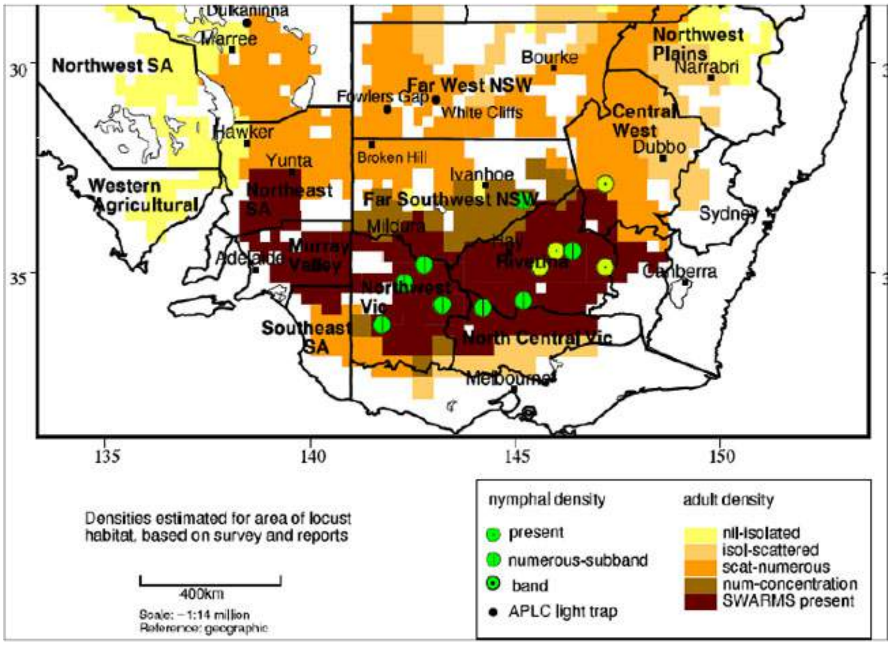


Figure 9: *Locust activity in south-eastern Australia in December 2010. Modified from DAFF (January 2011).*

Source: AgVic

Chortoicetes terminifera is a serious agricultural pest in Australia. An individual locust can consume 30–50% of its body weight daily, and swarms are very dense and highly mobile, migrating up to 700 km in a day. Swarms can be in excess of 50 individuals per square metre, consuming up to 10 tonnes of vegetation per day.

7.8.1 Effect on growing crops

While it is well known that cereals are particularly vulnerable to locusts, the susceptibility of the various pulses is uncertain, but we must assume that they could be attacked while they remain green. It is important to note that:

- Established green crops are susceptible to damage by adult locusts but tend to be avoided by hoppers (immature locusts), although crop edges can be damaged and may warrant a perimeter spray.
- Locusts cause little damage to crops that have dried off, but crops that are beginning to dry down when locusts begin to fly are still susceptible to attack.
- Even slight damage to pulse crops that have a high grain value or are destined for specific export markets could justify the cost of control (Photo 13).
- As a general rule, hopper and adult numbers should be closely monitored, and if any damage is seen, then spraying should be commenced immediately.
- Comply with label directions for the chosen insecticide and pay particular attention to WHP for harvest/windrowing or swathing, and for grazing/fodder. [38]

37 B Gagliardi, S Long, G Rose, L Golding, J Lieschke, T Daw Quadros, L Metzeling, V Pettigrove (2011) Australian plague locust response—Assessment of effects, Agriculture Victoria, Centre for Aquatic Pollution Identification and Management Technical Report #9 December 2011, http://agriculture.vic.gov.au/agriculture/pests-diseases-and-weeds/pest-insects-and-mites/plague-locusts/australian-plague-locust-reponse-assessment-of-effects

38 Pulse Australia (2015) Australian pulse bulletin: Impact of locusts on pulse crops and grain quality. http://pulseaus.com.au/growing-pulses/publications/locust-control

Photo 13: *Locust hatchlings cause the most damage when they form feeding bands that move across the land eating anything that is green.*

Source: Pulse Australia

7.8.2 Locusts can impact pulse deliveries

Key points:

- Locusts pose more than just a physical threat to pulse crop yields and quality.
- Controlling locusts before harvest is imperative to ensure marketable quality grain and to ensure successful delivery.
- Pulse growers need to make contact with their receival agent well in advance of harvest to discuss probable industry attitudes to high locust inclusion in the grain sample.
- Both receival agents and marketers may reject grain with high locust inclusion despite the sample technically meeting the receival standard for field insects.
- Grain staining, slimes and objectionable odours may arise from squashing live locusts during harvest. This material is difficult, if not impossible to remove.
- Objectionable material and odours in the sample will result in the product being rejected at the receival point.
- Only permitted chemicals are to be used for control of locusts.
- Maximum residue limits apply and grain samples may be collected and analysed for compliance with regulatory and market requirements. [39]

MORE INFORMATION

Pulse Australia: Locust can impact on pulse deliveries.

7.8.3 Management of locusts

The decision on how locusts in crops are best managed is affected by a range of factors including:

- Growth stage of the crop, i.e. is there any green plant material or has the crop completely dried off?
- Ability to harvest early—desiccation may be an option to advance harvest.
- Delivery standards required for the specific pulse—discuss requirements with potential buyers.
- Risk to market from pesticide residues—WHP for windrowing/swathing is the same as harvest.
- Ability to clean physical locust contamination from harvested grain.

Control

The most effective and easiest way for landholders to control locusts is by ground spraying the hoppers when they have formed into dense aggregations called bands (Table 5). This normally occurs from 1–2 weeks after hatching. The time available for

39 Pulse Australia (2010) Locusts can impact on pulse deliveries. Australian Pulse Bulletin PA 2010 #12, http://www.pulseaus.com.au/storage/app/media/crops/2010_APB-Pulse-locust-harvest.pdf

controlling an outbreak is short with hoppers taking about five weeks to develop into swarming adults. Hoppers are most likely to hatch in pasture paddocks and along roadsides, fence lines and non-cultivated ground around the crop perimeter, but some hoppers may hatch from egg beds laid within crops particularly bare areas such as wheel tracks where tram lining is practiced.

Table 5: *Summary of locust insecticide applications in Victoria on public land 2010–2011. 1 SC = suspensible concentrate, 2 ULV = ultra-low volume concentrate.*

Chemicals sprayed	# cases	Area treated (Ha)
Fipronil	30	488
Fenitrothion 1000	55	4,537
Green Guard SC 1	119	1,617
Green Guard ULV 2	5	3,370
Other	1	5
TOTAL	210	10,017

Source: AgVic

It is critical in these situations that the correct insecticide is used to avoid residue issues. Australian grain produce must meet minimum residue levels (MRLs). Individual deliveries of grain will be tested for chemical residues, to detect the use of unapproved pesticides and to ensure that WHPs have been followed.

- Only use an insecticide that is registered or has a permit for locust control in the specific pulse crop.
- Users must obtain, read and adhere to the conditions of APVMA permits prior to use.
- Follow label directions and pay attention to the WHP. Following pesticide application, the relevant withholding period MUST expire BEFORE cutting for hay, windrowing, harvest or undertaking of any similar operation.
- Plan well ahead in choosing the most appropriate product(s) to suit your situation as availability may become an issue as the season progresses.
- Be aware of the receival standards that apply to insect contamination (alive or dead) and grain damage from locust feeding.
- Be aware of nil tolerance for odour and taints that could arise from crushing locusts during harvest, handling or while in storage.
- Avoid inadvertent contamination of grain with other chemicals not used in pulses. [40]

In Victoria, growers are encouraged to report immature or adult locusts to the Victorian DEPI through the Customer Service Centre (136 186) or email plant.protection@depi.vic.gov.au. DEPI is particularly interested to receive any reports of egg-laying. In NSW, reports of egg-laying should be directed to the APLC**.**

7.9 Native budworm

The native budworm (*Helicoverpa punctigera* or, as it was known, *Heliothis punctigera*) is native to Australia and is distributed, particularly during spring, throughout much of the central and southern regions of the country. Native budworm is a major pest of pulse and canola crops in southern Australia and can develop large populations over extensive areas on native plants. Native budworm breeds over winter in the arid inland regions of South Australia, New South Wales, Queensland and Western Australia on desert plants before migrating into southern agricultural areas in late winter or spring causing damage to crops. They can migrate as far south as Tasmania. Migratory flights are unpredictable, as moths may be carried hundreds of kilometres from breeding areas by high altitude air currents.

MORE INFORMATION

Impact of locusts on pulse crops and grain quality.

Australia Plague Locust Response—Assessment of Effects.

40 Pulse Australia (2015) Australian pulse bulletin: Impact of locusts on pulse crops and grain quality. http://pulseaus.com.au/growing-pulses/publications/locust-control

Effective control requires understanding when the crop is at risk and the economic threshold for when to spray. In terms of production losses chickpeas, faba beans, tomatoes, field peas, lentils, lupins, canola and lucerne are important hosts. [41]

Unlike other parts of Australia or overseas where *Helicoverpa armigera* is abundant (the cotton bollworm or corn earworm), the control of native budworm poses no great problem in southern winter-growing areas of Australia. There is no known resistance to chemicals, and temperatures during the growing season do not allow a high level of activity until the crop is podding.

Lifecycles of *Helicoverpa* spp takes 4–6 weeks from egg to adult in summer and 8–12 weeks in spring or autumn. The lifecycle stages are egg, larvae, pupa and adult (moth) (Figure 10).

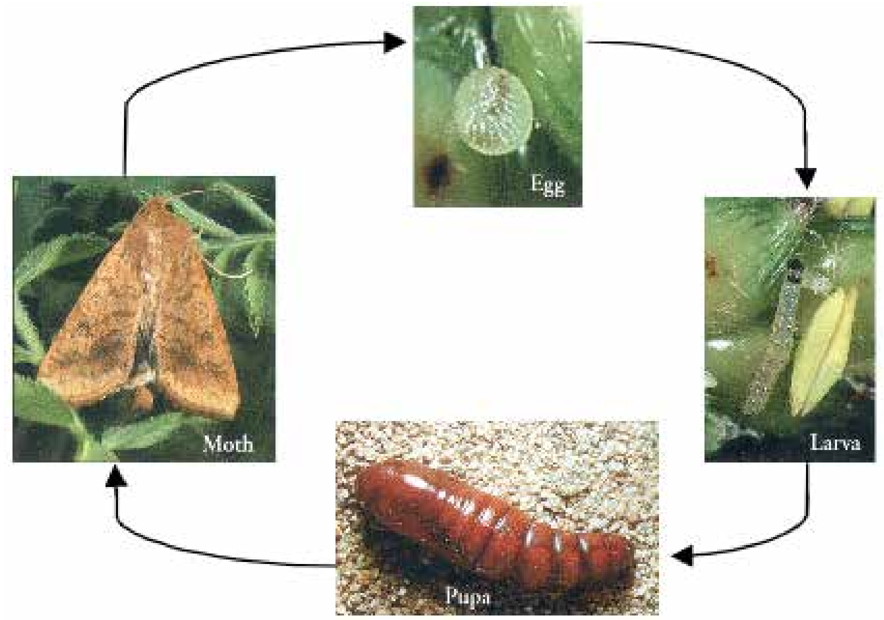


Figure 10: *Lifecycle of Helicoverpa.*

Source: Pulse Australia

Eggs

Budworm eggs can be found singly on the growing tips and buds of plants. They are small (about 0.5 mm in diameter) but quite visible to the naked eye after close inspection of the plant. Moths can lay up to 1,000 white spherical eggs mostly found near the top of the plant. They are white when first laid but they change colour to yellow and brown as they get closer to hatching, with larvae emerging in 7–14 days (Photo 14).

41 P Mangano, S Micic (2016) Management and economic thresholds for native budworm. DAFWA, https://www.agric.wa.gov.au/grains/management-and-economic-thresholds-native-budworm

Photo 14: *Left to right: fresh white, brown ring and black larval head in nearly hatching eggs.*

Source: DAFF.

Larvae/caterpillars

The newly hatched caterpillars (larvae) are very small and are often easily missed when inspecting a crop. When first hatched, they are about 1.5 mm long with dark brown heads and white bodies. The young caterpillars feed on leaf or pod material for about two weeks before they become large enough (5 mm long) to be noticed in the crop.

It takes a further four weeks until they are fully grown (40 mm long), which is about seven weeks from the time of egg-laying.

These development times are based on average spring temperatures when caterpillars are active in central cropping areas of Western Australia. Later in the season, or in more northerly areas, developmental rates for caterpillars will be faster.

The caterpillars vary greatly in colour from green through orange to dark brown and are often seen with their heads inside pods. They usually have dark stripes along the body and are sparsely covered with fine bristles. [42]

During full development they will pass through six or seven growth stages or instars, until they are 35–40 mm long (Figure 11). When fully grown, their colour ranges from green, yellow, buff, red or brown to almost black, with a broad yellow-white stripe down each side of the body and a dark stripe down the centre of the back. The skin of the caterpillars appears rough to touch, due to long, dark hairs on prominent bumps on the body surface (Figures 12 and 13). New moth flights and egg-laying will result in caterpillars of varying sizes in a crop. Caterpillars eat increasing quantities of seed and plant material as they grow with the last two growths stages (fifth and sixth instar) responsible for eating over 90% of their total grain consumption.

42 P Mangano, S Micic (2016) Management and economic thresholds for native budworm. DAFWA, https://www.agric.wa.gov.au/grains/management-and-economic-thresholds-native-budworm

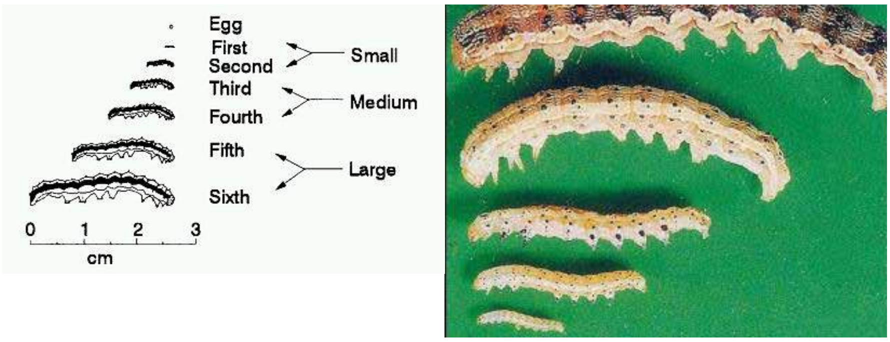


Figure 11: *Approximate instar sizes of the budworm.*

Source: Agriculture Victoria, DAFWA

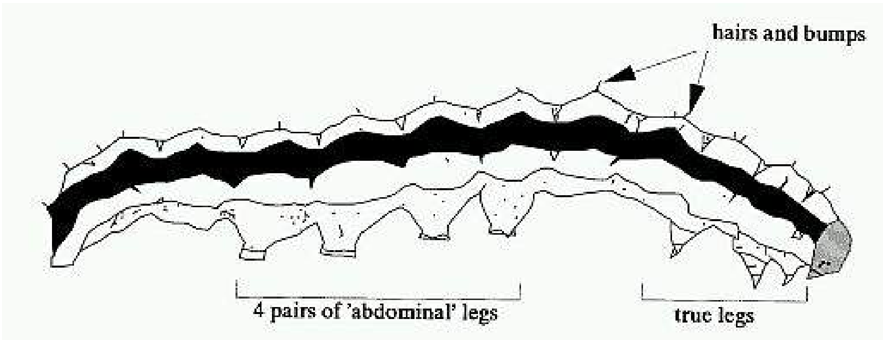


Figure 12: *Distinguishable features of native budworm larva.*

Source: Agriculture Victoria

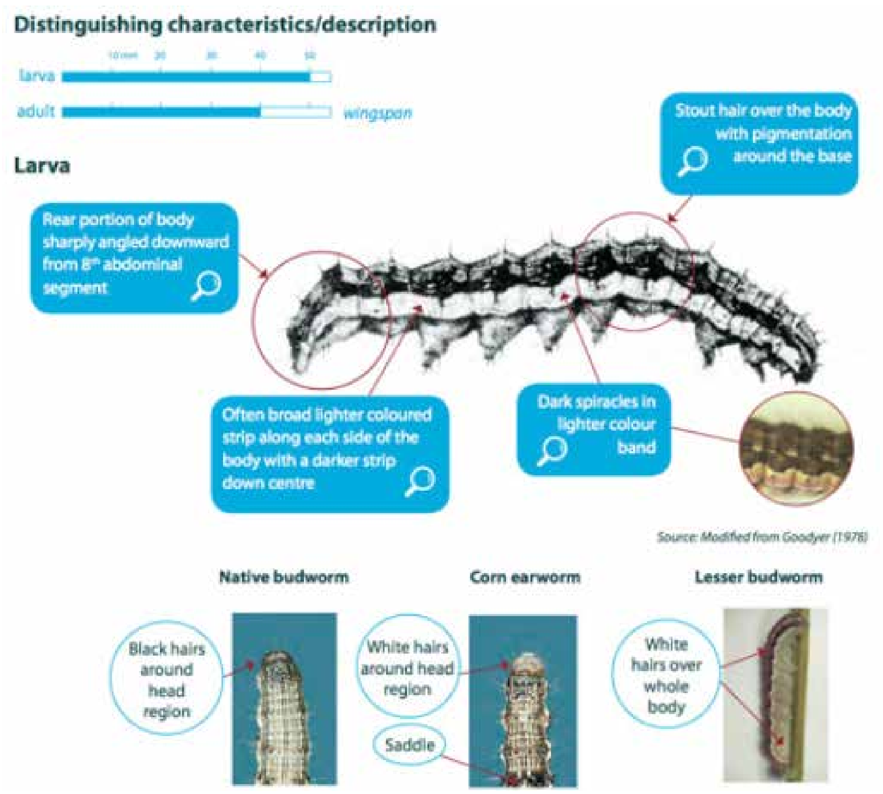


Figure 13: *Distinguishing characteristics/description of native budworm.*

Source: Bellati *et al.* 2012 in cesar

Pupa

When fully mature, the caterpillars crawl to the ground, burrow from 20–150 mm in depth into the soil and pupate. Pupae are cigar-shaped, 12–22 mm long, and during development change in colour from a yellow-orange to a shiny dark brown. The length of the pupal stage depends on several environmental factors and varies from two weeks to several months.

Adult

Adult moths are medium sized (wingspan 30–40 mm) and stout bodied (Photos 15 and 16). The forewings are buff-olive to red-brown with numerous dark spots and blotches. The hind wings are pale grey with dark veins and a dark band along the lower edge. Moths are usually active during the evening and night. Moths can be seen in crops by their rapid, low-level flight that takes a zigzag path then diving into a crop to lay eggs.

Photo 15: *Native budworm larvae showing prominent hairs (left) and buff coloured adult (right).*

Source: cesar

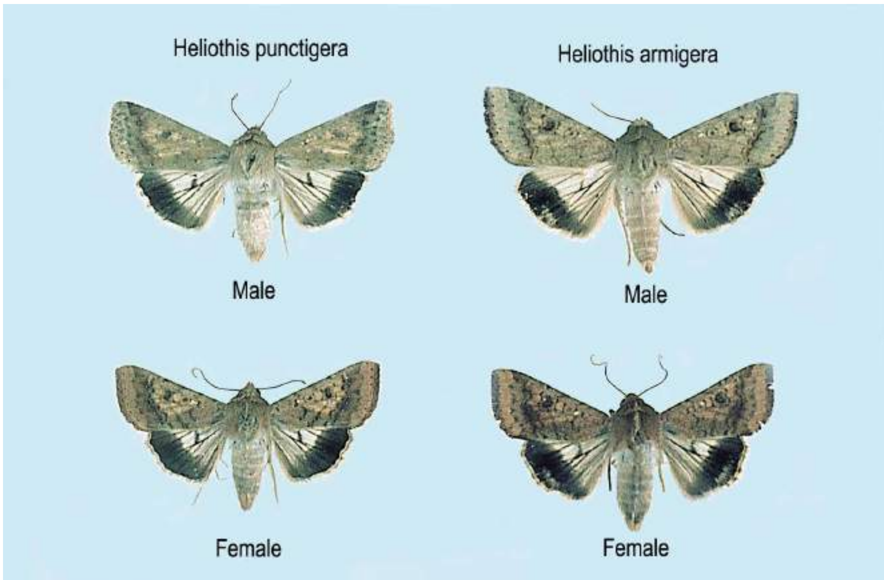


Photo 16: *Helicoverpa punctigera male (top left) and female (bottom left) compared to Helicoverpa armigera male (top right) and female (bottom right). Both male and female Helicoverpa armigera have a white patch on their hindwings.*

Source: GRDC

Mortality of eggs and caterpillars

Only a small proportion of eggs laid by moths survive to the damaging large caterpillar stage. There is a high death rate in the egg and small caterpillar stages. Eggs may be dislodged and small caterpillars may become stuck or drown due to wet weather.

Predators and parasitoids also affect population numbers and can suppress them below economic damage thresholds. If numbers of native budworm are high, populations can crash due to viruses and disease. [43]

7.9.1 Varietal resistance or tolerance

Crops vary in their attractiveness to moths as sites for egg-laying. Crop density and crop growth stage (flowering and podding) will affect the number of eggs laid by the native budworm moths. The feeding behaviour of caterpillars also changes according to the type of crop the caterpillars are feeding upon.

Chickpea, field pea, lentil and faba bean crops:

- These crops are very susceptible to all sizes of caterpillars during the formation and development of pods.
- Tiny caterpillars can enter emerging pods and damage developing seed or devour the entire contents of the pod.

7.9.2 Damage caused by pest

Native budworm caterpillars most frequently attack the fruiting parts of plants (Photo 17) but also feed on the terminal growth, flowers and leaves. All pulse crops grown in southern Australia are susceptible to attack, especially when pods are present. This includes chickpea, field pea, faba bean, lentil, lupin and canola. Direct losses are usually associated with yield through grain being wholly or partially consumed by the caterpillar. However indirect losses from quality issues can result in downgrading or even rejection from high levels of damaged grain, weathering or fungal infections from holes in pods, discolouration or odour. [44] A single caterpillar can attack four to five pods before reaching maturity and will feed in crops for four to six weeks.

Photo 17: *Large Helicoverpa larva feeding on a chickpea pod.*

43 P Mangano, S Micic (2016) Management and economic thresholds for native budworm. DAFWA, https://www.agric.wa.gov.au/grains/management-and-economic-thresholds-native-budworm

44 T Bray (2010) Managing native budworm in pulse crops. Pulse Australia, Southern Pulse Bulletin PA 2010 #18, http://www.pulseaus.com.au/storage/app/media/crops/2010_SPB-Pulses-native-budworm.pdf

Cost of native budworm

Losses attributed to native budworm come from direct weight loss through seeds being wholly or partly eaten. Grain quality may also be downgraded through unacceptable levels of chewed grain or fungal infections introduced via caterpillar entry into pods. The percentage of broken, chewed and defective seed found in grain samples affects the final price of pulse crops marketed for human consumption. This applies particularly to the large-seeded crops such as kabuli chickpea, faba bean, lentil and field pea. [45]

7.9.3 Conditions favouring development

Native budworm is the major pest of grain legumes in spring (Table 6). At this time of year, the adult moths fly from inland breeding areas, on weather systems, and lay eggs in pulse and canola crops. Early spring is the time when grain growers should be checking all pulse and canola crops for native budworm eggs and larvae as crops reach the susceptible flowering and podding stages.

Table 6: *Conditions leading to risk of damage and loss from native budworm in chickpeas.*

High risk	Moderate risk	Low risk
Wet winters in breeding areas of central Australia + suitable weather conditions that bring moths from the north on northerly winds, resulting in spring migrations. Repeated influxes of moths over long periods, resulting in need for continuous monitoring and potentially repeat infestations.	Broadleaf weeds hosting cutworm and Helicoverpa that move into the crop as large, damaging larvae. Hot weather in spring can cause small larvae to burrow into pods. Wet harvest weather resulting in pods that are 'softer' for longer and susceptible to damage right up to harvest.	Dry winters in breeding areas. Low source population. Absence of frontal wind systems that provide opportunities for migration.

Source: IPM Guidelines

Species composition in the crop will be influenced by a number of factors, such as:

- Winter rainfall in inland Australia, which drives populations of *H. punctigera*, and the occurrence and timing of wind systems that carry *H. punctigera*.
- Relative timing of flowering–podding (attractive and susceptible) stages and the immigration of *H. punctigera* and emergence of *H. armigera* from overwintering diapause.
- Geographic location. In temperate regions, most of the *H. armigera* population overwinters from mid-March onwards and emerges during September–October. *Helicoverpa punctigera* is usually the dominant species through September when moths are migrating into eastern cropping regions. Seasonal variation can lead to *H. armigera*-dominant early infestations in some years. [46]

Native budworm breeds during winter in the semi-arid regions of Australia and moths can often migrate long distances in early spring into the southern agricultural districts. Adults are nocturnal and often seen around lights at night which can be an indicator of their arrival.

Crop types can vary in attractiveness to moths for egg-laying and as a general rule lentils are the most attractive followed by field pea, vetch, faba bean, chickpea, and lupin. Crop health, density and growth stages (flowering and podding) will generally affect the number of eggs laid with moths mostly preferring the more advanced

45 P Mangano, S Micic (2016) Management and economic thresholds for native budworm. DAFWA, https://www.agric.wa.gov.au/grains/management-and-economic-thresholds-native-budworm

46 M Miles (2013) Chickpea insect pest management. Department of Agriculture, Fisheries and Forestry, Queensland, http://ipmworkshops.com.au/wp-content/uploads/Chickpea_IPM-Workshops_north-March2013.pdf

dense and succulent areas. Feeding behaviour of caterpillars also changes according to the pulse type with field pea, chickpea, lentil and faba bean crops being more susceptible to all sizes of caterpillars during pod formation and development. [47]

7.9.4 Thresholds for control

Thresholds depend on crop value, cost of control and tolerance of feeding damage. Suggested thresholds (check 5–10 sites) are:

- chickpeas (kabuli): 2–3 larvae in 10 sweeps
- chickpea (Desi type): 5 larvae in 10 sweeps

Threshold tables

The losses given (Table 6) are for the number of caterpillars netted in crops during early pod formation for all crops except lupins and canola. For these canola and lupins numbers are during pod maturation.

To use the table you need to substitute:

- control costs with your own actual costs
- expected grain price per hectare based

This will calculate the economic threshold or the number of caterpillars that will cause more financial loss than the cost of spraying (e.g. Table 7).

Table 7: *Example to calculate the economic threshold or the number of caterpillars that will cause more financial loss than the cost of spraying.*

The on-farm value of field peas is $185 per tonne (t)
The cost of control is $12 per hectare (ha)

ET = C ÷ (K x P)
Where:
ET = Economic threshold (numbers of grubs in 10 sweeps)
C = Control cost (includes price of chemical + application) ($ per ha)
K = Kilogram per hectare (ha) eaten for every one caterpillar netted in 10 sweeps or per square metre (see Table 2)
P = Price of grain per kg (price per tonne ÷ 1000)

Therefore economic threshold for field pea = 12 ÷ (50 x (185 ÷ 1000)) = 1.3 grubs per 10 sweeps

Source: DAFWA

Table 8: *Economic thresholds (ET) for native budworm on chickpeas.*

P Grain price per tonne	C Control costs including chemical + application	K Loss for each grub in 10 sweeps (kg/ha/grub)	ET Grubs in 10 sweeps	ET Grubs in 5 lots of 10 sweeps	ET Grubs (>15 mm) per m^2
420	10	30	0.8	4	-

Note: Growers using this table to calculate spray thresholds should substitute their own control costs and the current on-farm grain price expected.

Source: DAFWA

Where:

ET = Economic threshold (numbers of grubs in 10 sweeps)
C = Control cost (includes price of chemical + application) ($ per ha)
K = Kilogram per hectare (ha) eaten for every one caterpillar netted in 10 sweeps or

47 T Bray (2010) Managing native budworm in pulse crops. Pulse Australia, Southern Pulse Bulletin PA 2010 #18, http://www.pulseaus.com.au/storage/app/media/crops/2010_SPB-Pulses-native-budworm.pdf

per square metre
P = Price of grain per kg (price per tonne ÷ 1000).

The ready-reckoner table (Table 9) shows data for a range of larval densities, and crop prices. Putting a dollar value on the predicted yield loss if nothing is done to control the *Helicoverpa* infestation is a useful way to assess the economic benefit (or otherwise) of spraying.

Table 9: *The value of yield loss ($/ha) caused by Helicoverpa larvae in chickpea for a range of larval densities (determined by beat sheet sampling) and grain prices. NOTE: Control is warranted if the cost of control is less than the value of the yield loss predicted.* [48]

Chickpea price ($/t)	Value of yield loss ($/ha)				
	1 larva/m^2	2 larva/m^2	3 larva/m^2	4 larva/m^2	5 larva/m^2
200	4	8	12	16	20
300	6	12	18	24	30
400	8	16	24	32	40
500	10	20	30	40	50
600	12	24	36	48	60

In Figure 14, the field has an average of 4.2 larvae per m^2 (adjusted for mortality of small larvae). Assuming a chickpea price of $400/t, the table of potential yield loss (refer to Table 5) shows the cost of not controlling to be $32/ha. In this example, if the cost of control is less than $32/ha then it is economic to spray.

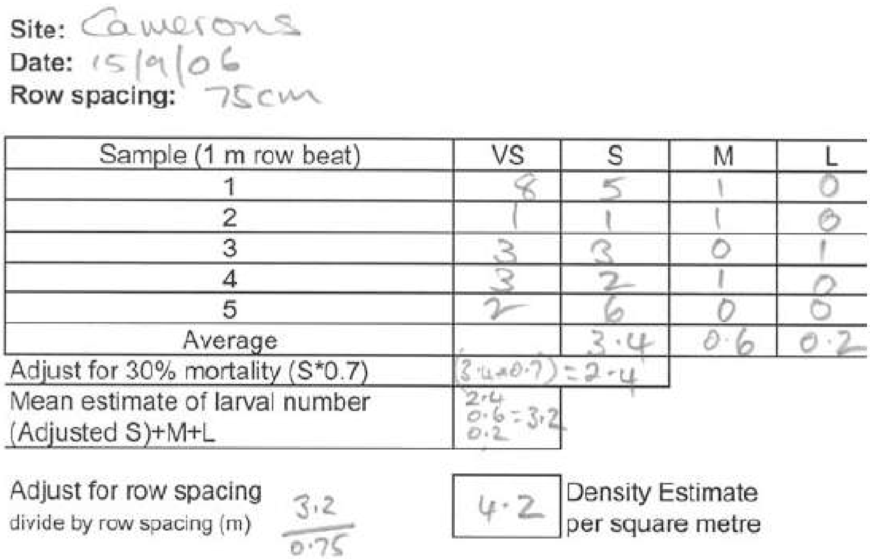


Figure 14: *Example of a field check sheet with sampling data recorded for Helicoverpa larvae in chickpea.*

Source: DAFF

Adjusting thresholds

Use of the table and calculations will provide a personalised and more precise measure of potential loss from native budworm damage. Sometimes the loss would turn out to be less than predicted, if, for example, the season is shortened by a lack of moisture.

48 M Miles (2013) Chickpea insect pest management. Department of Agriculture, Fisheries and Forestry, Queensland, http://ipmworkshops.com.au/wp-content/uploads/Chickpea_IPM-Workshops_north-March2013.pdf

Premiums paid for exceeding quality standards for high value and large-seeded pulses (like kabuli chickpea) may necessitate even lower thresholds than those provided in the table. [49]

7.9.5 Making a decision to control

Several factors (in addition to number of larvae) will influence a decision on whether to spray, timing and product choice:

- Age structure of the larval population may need to be considered in relation to time to desiccation or harvest. For example, a late egg-lay is unlikely to result in economic damage if the crop is 7–10 days away from harvest.
- Proportions of *H. punctigera* making up the total population are important and can be determined by visual identification, time of year, pheromone trap catches and local experience.
- Spray conditions and drift risk must be considered.
- Information on insecticide options, resistance levels for *Helicoverpa* and recent spray results in the local area should be sought.
- Residual of the products may have implications.

Selecting control options

We depend on insecticides for the management of *Helicoverpa* in chickpeas, and the high usage of a limited group of compounds against successive pest generations imposes severe selection pressure. Invariably, selection is for individuals in a population that are not killed by normal application rates of insecticides. With continued insecticide application, the frequency of resistant individuals in the population increases, leading to field-control failures.

The potential for natural enemies of *Helicoverpa* (predators, pathogens and parasitoids) to limit the development of damaging populations of larvae, while typically low in chickpeas, may also influence product selection.

'Spray small or spray fail'

Spraying should be carried out promptly once the threshold has been exceeded. Insects grow rapidly under warm spring conditions, and a few days' delay in spraying can result in major crop damage and increased difficulty in control..

If a spray application is delayed for more than 2 days, for any reason, the crop should be rechecked and reassessed before any control action is implemented. [50]

7.9.6 Management of Helicoverpa

Monitoring

All crops should be scouted weekly during flowering for moth activity and eggs, then at least two times per week during pod-fill for eggs and larvae. The main egg-laying period is often around the flowering period when moths can be quite abundant. Eggs can often be found on the vegetative or floral growing points, new leaves, stems, flowers, flower buds and young pods. They may not be obvious to the untrained eye unless there is a heavy egg-lay or until small larvae can be found. [51]

Light traps and pheromone traps can indicate presence of adults in spring. Monitor crops 1–2 weekly until pod-set, then increase frequency when moths and/or larvae are detected.

49 P Mangano, S Micic (2016) Management and economic thresholds for native budworm. DAFWA, https://www.agric.wa.gov.au/grains/management-and-economic-thresholds-native-budworm

50 M Miles (2013) Chickpea insect pest management. Department of Agriculture, Fisheries and Forestry, Queensland, http://ipmworkshops.com.au/wp-content/uploads/Chickpea_IPM-Workshops_north-March2013.pdf

51 Bray T (2010) Managing native budworm in pulse crops. Pulse Australia, Southern Pulse Bulletin PA 2010 #18, http://www.pulseaus.com.au/storage/app/media/crops/2010_SPB-Pulses-native-budworm.pdf

- Use beat sheet (wide rows), sweep net (narrow rows) and/or visual sampling (Photo 18). Where border sprays were used to control pea weevil, start sampling well into the crop.
- Look for eggs—on leaves, buds and flowers. Start looking for eggs mid-August or when the crops start flowering and moths are detected.
- Repeat sampling at 5–10 sites across the field—more sites give greater level of confidence.
- Assess pod burrowing by looking for holes and splitting open 20–40 pods and look for larval damage.
- Record number, sizes, and crop development stage. Record /m^2 to compare numbers with appropriate thresholds. In southern grain regions, average number of caterpillars per 10 sweeps of an insect sweep net is the standard for comparison and thresholds.

The quickest and easiest method to sample most crops is to sweep with an insect net, taking 2 m long sweeping arcs, using the standard net size (380 mm diameter).

Multiples of 10 sweeps should be taken in several parts of the crop. If more than 10 consecutive sweeps are made, there are likely to be too many dead flowers and leaves in the sample to locate the small caterpillars easily.

Netting is most efficient in short and thin crops and less efficient in tall dense crops. It is very important to keep the lower leading edge of the sweep net slightly forward of the net opening so that dislodged grubs are picked up and carried into the net.

Photo 18: *Sweep-netting a chickpea crop (left) and use of a beat sheet (right).*

Source: DAFWA and The Beatsheet

In years of large moth influxes or wet springs where crops continue to flower/pod, monitoring should continue until pods are dry and no longer able to be penetrated by larvae.

How to use a beat sheet

Place the beat sheet with one edge at the base of a row. On 1-m row spacing, spread the sheet out across the inter-row space and up against the base of the next row. Draping over the adjacent row may be useful for row spacing <1 m, or where there is canopy closure. It also minimises the chance of larvae being thrown off the far side of the sheet. With a 1-m-long stick (dowel, heavy conduit), shake the row vigorously 10 times to dislodge larvae from the plants. Measure and count larvae on the sheet. A standard beat sheet is made from plastic or tarpaulin material with heavy dowel on each end to weigh down the sheet. The beat sheet is typically 1.3 m wide by 1.5 m long. The extra 0.15 m on each side catches insects thrown out sideways.

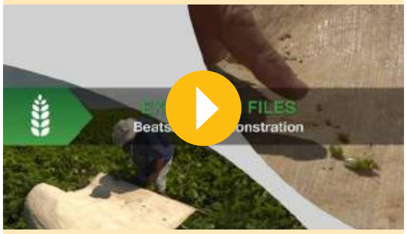
Using a sweep net to monitor Helicoverpa

A standard sweep net has a cloth bag and an aluminium handle. With heavy use, the aluminium handle can shear off; more robust, wooden handles are often fitted by agronomists.

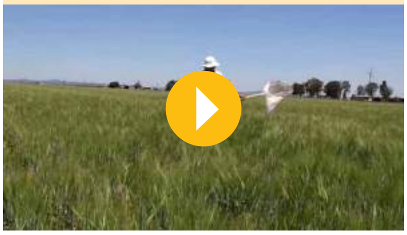
Where crops are sown on narrow row spacings and it is not possible to get a beat sheet between the rows, a sweep net can be used to sample *Helicoverpa*. Hold the sweep net handle in both hands and sweep it across in front of your body in a 180° arc. Take a step with each sweep. Keep the head of the net upright so the bottom

VIDEOS

3. GCTV16: Extension files – IPM Beatsheet Demo.



4. How to use a sweep net to sample for insect pests.



of the hoop travels through the canopy. Use sufficient force in the sweep to pass the hoop through the canopy and dislodge larvae. Take 10 sweeps and then stop and check the net for larvae. Record the number and size of larvae in each set of 10 sweeps. Repeat at additional sites across the field. [52]

Monitoring for adult moths

Male moths are easily captured in pheromone (female sex scent) traps. Pheromone traps can provide an indication of the species presence in a region but are not a reliable predictor of actual egg-lay or grub numbers within a crop (Photo 19).

Photo 19: *Pheromone trap used for detecting moths.*

Source: Pulse Australia

Pheromone trap catches—data updates

Stay up to date with native budworm numbers in your local areas. Weekly trap catch data for *H. punctigera* and *H. armigera* from locations across all states can now be viewed online. The adjustable bar below the map allows selection of a time period (1 wk, 2 wks, 1 mth etc.). [53]

Recording of monitoring data for decision-making

Keeping records should be a routine part of insect checking. Successive records of crop inspections will show whether pest numbers are increasing or decreasing, and will help in deciding whether a control is necessary.

Records of insect checking should include as a minimum:

- date and time of day
- crop growth stage
- average number of pests detected, and their stage of development
- checking method used and number of samples taken

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52 M Miles (2013) Chickpea insect pest management. Department of Agriculture, Fisheries and Forestry, Queensland, http://ipmworkshops.com.au/wp-content/uploads/Chickpea_IPM-Workshops_north-March2013.pdf

53 The Beatsheet. https://jamesmaino.shinyapps.io/MothTrapVis/

- management recommendation (economic threshold calculation)
- post-spray counts

The *Helicoverpa* size chart (Table 10) is an essential reference for decision-making, particularly in chickpea where larval size is taken into account in the economic threshold (beat sheet threshold), and is important in ensuring that any control action is well targeted against susceptible larvae.

Eggs and very small larvae are not included in the economic threshold for *Helicoverpa* (beat sheet threshold) due to high natural mortality.

Table 10: *Helicoverpa larval size categories and actual sizes.*

Helicoverpa larval size categories and actual sizes		
Actual larval size	**Larval length (mm)**	**Size category**
	1-2 mm	very small
	4-7	small
	8-23	medium
	24-30+	large

Source: IPM Guidelines for Chickpeas

Chemical control

Key points:

- Aim to control larvae before they enter the protection of pods (Photo 20)—target small larvae <7 mm.
- Synthetic pyrethroids are very effective but their broad-spectrum activity has a negative impact on any beneficial insects present.
- Commercially available NPV gives up to 80% control in chickpeas.
- Bt is effective against *Helicoverpa*.
- There is usually a range of rates on the insecticide label to allow for varying conditions such as the size of the caterpillars. The choice of rate should not be solely driven by the lowest price. Also consider impact of chemical use on other pests and beneficial species.
- Inspect crops after spraying to ensure chemical applications have been effective and to detect further infestations until the crop is no longer susceptible. [54]

54 IPM Guidelines (2016) Native budworm in winter pulses. GRDC, http://ipmguidelinesforgrains.com.au/pests/helicoverpa/native-budworm-in-winter-pulses/

Photo 20: *Helicoverpa entering a chickpea pod.*

Source: Pulse Australia

The decision to spray for chickpea needs to be considered from the time of first podding.

If caterpillar numbers are below the threshold levels provided, the decision to spray should be delayed and periodic sampling continued. One well-timed spray to control native budworm caterpillars should be sufficient in most situations.

Sweep netting of the crop should be carried out after spraying to confirm that the required level of control has been obtained. Effectively applied synthetic pyrethroids will prevent reinfestation for up to six weeks after spraying. Subsequent caterpillar hatchings will usually be too late to cause any damage. [55]

One well-timed spray with a synthetic pyrethroid should provide good control of larvae and prevent reinfestation for up to six weeks. Synthetic pyrethroid insecticides are very effective but their broad-spectrum activity impacts negatively on beneficial insects.

Refer to the beneficial impact table for more information.

Most insecticides used for budworm control are contact chemicals and therefore it is important to have complete coverage of the target. A medium spray quality is necessary as they are light and will flow around with the airstream, particularly at low airflow rates and increase the chance of striking the target inside the canopy.

They are however more subject to off-target drift when conditions are not ideal and air induction/inclusion or venturi nozzles are becoming more popular to achieve more uniform droplet size at low pressures. Most insecticide spraying should be at less than 210 µm (microns) VMD (volume mean diameter, i.e. 50% of droplets >2 10µm, and 50% of droplets <210 µm). This is generally finer than for most herbicides and labels will sometimes specify a droplet range along with water volumes. [56]

There are several insecticides registered for the control of native budworm. Timing and coverage are both critical to achieving good control. Try to target small larvae up to 7 mm in length and apply insecticides before larvae move into flowering pods.

55 P Mangano, S Micic (2016) Management and economic thresholds for native budworm. DAFWA, https://www.agric.wa.gov.au/grains/management-and-economic-thresholds-native-budworm

56 Bray T (2010) Managing native budworm in pulse crops. Pulse Australia, Southern Pulse Bulletin PA 2010 #18, http://www.pulseaus.com.au/storage/app/media/crops/2010_SPB-Pulses-native-budworm.pdf

IPM options include the use of Bt (*Bacillus thuringiensis*) and Nuclear polyhedrosis virus (NPV) based biological insecticides. Small larvae are generally easier to control because they are more susceptible to insecticides, and leaf feeding makes them susceptible to ingestion of active residues on the plant surface. Larvae entrenched in buds and pods will be more difficult to control and chemical residual will be important in contacting them.

The crop should be re-inspected 2–4 days after spraying to ensure enough caterpillars have been killed to prevent future damage and economic loss. In years of very high moth activity and extended egg lays, a second spray may be required. [57] Remember that larger larvae will often burrow into pods and control becomes more difficult being shielded from the spray and more likely to survive.

Be aware of insecticide withholding periods (WHP) close to harvest and remember that windrowing is classified as harvest.

Many traditional pyrethroid insecticides (e.g. alpha-cypermethrin) have a 21-day WHP in canola; however there are newer generation pyrethroids available (e.g. Trojan® or Karate®) with a shorter 7-day WHP. [58]

Biological control

A key component to any IPM is to maximise the number of beneficial organisms and incorporate management strategies that reduce the need for pesticides. Correct identification and monitoring is the key when checking for build-up or decline in beneficials. There are many natural enemies that attack native budworm. The egg stage is susceptible to the parasite Trichogramma ivalae, a minute wasp that has been recorded in up to 60% of eggs along with egg predators such as ladybird beetles, lacewings and spiders. Beneficials attacking larvae include shield bugs, damsel bugs, assassin bugs, tachinid flies (their larvae prey on caterpillars), orange caterpillar parasite, two-toned caterpillar parasite, orchid dupe, lacewings and spiders. Naturally occurring fungal diseases and viruses also play an important role in some seasons.

Beneficials:

- Be aware of key beneficials before larvae infest the crop.
- *Trichogramma* wasps parasitise *Helicoverpa* eggs and *Microplitis*, *Heteropelma*, *Netelia* sp. and other wasps parasitise *Helicoverpa* larvae.
- Predatory bugs such as *Geocorris* and *Nabis* prey on eggs and small larvae while *Cermatulus* and *Oechalia* also attack larger larvae.
- Ants and spiders also eat *Helicoverpa* eggs and larvae.
- NPV is a virus which only infects *Helicoverpa* species. This occurs naturally but can also be applied—see chemical control. [59]

7.9.7 Management nearing desiccation and harvest

Hot, dry weather will rapidly advance a chickpea crop (Photo 21) which means that very small and small larvae are unlikely to survive on leaves of rapidly deteriorating quality. As the pods dry they also become more resistant to damage by small to medium larvae. In summary, this means that the major source of damage in a senescing crop is late and by medium and large larvae.

57 G McDonald (2015) Native budworm. cesar February 2015, http://www.cesaraustralia.com/sustainable-agriculture/pestnotes/insect/Native-budworm

58 GRDC (2014) Budworm in Western Australia. https://grdc.com.au/Media-Centre/Hot-Topics/Budworm-in-Western-Australia

59 IPM Guidelines (2016) Native budworm in winter pulses. GRDC, http://ipmguidelinesforgrains.com.au/pests/helicoverpa/native-budworm-in-winter-pulses/

Photo 21: *Chickpea crop nearing desiccation and harvest.*

Source: The Beatsheet

Therefore, the recommended approach to managing *Helicoverpa* populations in the later stages of a chickpea crop is to continue to monitor both number and size of larvae. If the population of medium and large larvae exceeds the economic threshold, AND the crop is still susceptible then treatment may be warranted.

At this stage of the crop, a wait and see approach (continue checking the crop 1–2 times a week) to is recommended principally because it is difficult to predict a week or two ahead how fast a crop will dry down, and what the *Helicoverpa* population will be while the crop is still susceptible. The alternative approach is to treat above threshold populations of small larvae when they are detected. This approach is likely to result in treatment of fields that subsequently would not have been at risk of damage, particularly if the crop dries faster, or larval mortality is higher than expected.

The options available for the treatment of *Helicoverpa* infestations late are limited because of WHP. Methomyl has a 1 day WHP while thiodicarb has a 21 day WHP. Indoxacarb (Steward™) has a 21 day WHP, but no more than one application is permitted per crop growth cycle. Check with others in your local area on their experience with the efficacy of options when making a choice. [60]

7.9.8 Broader management considerations

Close monitoring can pay off. In many cases, the larval infestation may not progress past the 'small' stage, and therefore, control is unwarranted. Regular close checking, and reference to records from successive checks, will enable you to determine larval survival.

Aim for one well-timed spray. Chickpea can tolerate moderate to high numbers of *Helicoverpa* larvae (10–20 larvae/m^2) through late-vegetative and early-flowering growth stages. However, agronomists may suggest that numbers this high during flowering would warrant immediate spraying. Even with mortality, an economic threshold may be exceeded as soon as pod-set begins. This situation potentially leads to high numbers of advanced stage larvae, resulting in more costly and less reliable control.

60 The Beatsheet (2008) Managing Helicoverpa larvae in chickpea crops close to desiccation and harvest. The Beatsheet 05 November 2008, http://thebeatsheet.com.au/managing-helicoverpa-larvae-in-chickpea-crops-close-to-dessication-and-harvest/

Most yield loss will be sustained from damage caused during pod-fill, and this is the most critical stage for crop protection. Larval infestations are likely to be of mixed ages by the time the crop is well into podding. Products such as Steward™ and Larvin® will adequately control a wide range of larval sizes, and offer around 10–14 days of residual protection if applied to plants that are not actively growing. [61]

7.9.9 Native budworm in failed chickpea crop—subsequent threats

Helicoverpa larvae surviving in failed chickpea crops may be a threat to the following crop. With some chickpea crops being sprayed out, rather than harvested, there are reports of *Helicoverpa* larvae surviving on crop residues. The survival of larvae, particularly large late instar larvae, poses a threat to subsequent crops.

Larvae will persist in what may appear to be dead chickpea plants, surviving by feeding on the stems, or any other green bits that may take longer to dry down. Clearly, cool wet weather will slow the rate of crop dry down, and may allow the larvae to survive longer. Emerging seedlings are very susceptible to *Helicoverpa* feeding.

While the use of herbicides is not recommended for the control of insect pests, herbicides may have some impact on *Helicoverpa* larvae. In the late 1990s, it was observed that Sprayseed® applied to finish off chickpea trap crops also killed *Helicoverpa* larvae, but such observations need to be confirmed with further data. There are no observations on the impact of other herbicides on *Helicoverpa*, so it remains important to check the crop residues. Please report any suspected larval deaths (due to herbicides) to the Beatsheet.

If the chickpea field is going into fallow, or not being planted immediately, the larvae will likely starve to death, or complete their development and pupate. [62]

7.9.10 Checking compatibility of products used in mixtures

With the fungicide spray programs recommended for Ascochyta blight control, mixing of fungicides with insecticides is becoming more common. However, some product formulations are NOT compatible with available fungicides.

Check compatibility of potential mixing partners before recommending and applying.

Always read the label supplied with each product before use.

Compatibility of insecticides with mancozeb formulations

It is the responsibility of the agronomist ultimately to ensure that any recommendation is safe for the crop.

Table 11 outlines some considerations when using chlorothalonil within 10 days of an insecticide application. These lists are by no means exhaustive and have been compiled using current, available data from the chemical.

Always check with individual companies and read product labels for specific information.

Note that formulations can vary between companies or they may be changed without notice. Compatibilities provided are a guide only and should be followed up with companies if problems occur.

Always read the label supplied with the product before each use.

61 M Miles (2013) Chickpea insect pest management. Department of Agriculture, Fisheries and Forestry, Queensland, http://ipmworkshops.com.au/wp-content/uploads/Chickpea_IPM-Workshops_north-March2013.pdf

62 The Beatsheet (2010) Helicoverpa larvae surviving in failed chickpea crops may be a threat to the following crop. The Beatsheet 08 November 2010, http://thebeatsheet.com.au/helicoverpa-larvae-surviving-in-failed-chickpea-crops-may-be-a-threat-to-the-following-crop/

Always ensure that a product (or mixture) is safe for the crop before recommending and applying. [63]

Table 11: *Compatibilities of various insecticides with chlorothalonil.*

Product	Chlorothalonil compatibility	Considerations
Steward™ (indoxacarb)	Yes. Widely used with chlorothalonil and no know compatibility issues	
Oil-based emulsifiable or flowable pesticides	Some incompatibilities. The excerpt (right) is from the Crop Care Barrack 720 label. Also see labels of other chlorothalonil products available under permit.	DO NOT tank mix Crop Care Barrack 720 with EC formulations when spraying after shuck fall. COMPATIBILITY: This product is compatible with wettable powderformulations of the most commonly used fungicides, insectides and miticides. Do not combine with oil-based emulsifiable or flowable pesticides, unless prior experience has shown the combination to be physically compatible and non-injurous to your crop. This product should not be mixed with spraying oils or sprayed on to crops that have been sprayed with oil for at least 10 days after the oil spray. Oils should not be sprayed on crops treated with this product for at least 10 days after the last spray. Wetting agents have not improved performance. Under some conditions, certain surfactants may cause injury.

Source: compiled with the assistance of Bayer CropScience, Sumitomo Chemical Australia, DuPont, Crop Care Australasia and Infopest

7.9.11 Post-spray assessments

After applying a spray to control a pest infestation, a post-spray assessment or follow-up check is essential to ensure that pest numbers were successfully reduced to below the threshold.

Sometimes sprays fail to work as effectively as required or expected. This can occur for a variety of reasons, such as inadequate application (coverage, timing), insecticide resistance, or too-high expectations of the product selected. Poor application is sometimes mistaken as resistance.

Where a spray failure is suspected, detailed records can assist in determining the cause of the apparent failure.

With products such as Steward™, the phenomenon of growth dilution is often evident in chickpeas. That is, the growth that was present at the time of application may still have residual activity from the insecticide but new growth will not. It has been observed that small larvae can feed on this new growth but incur no crop damage. Rechecking fields sprayed with Steward™ or Larvin® can be complicated and will require regular assessment.

Record spray decision and re-check to confirm control success or failure. Record details of application equipment (nozzle size etc.) as well as time of day and weather conditions. This may help interpret what might have gone wrong where poor control is achieved. [64]

63 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

64 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

Nematode management

Key messages

- Nematodes are common soil pests that feed on the roots of a wide range of crop plants, irrespective of soil type and rainfall.
- Root-lesion nematodes (RLN) reduce development of lateral roots, which decreases the ability of plants to extract water and nutrients.
- *Pratylenchus neglectus* and *Pratylenchus thornei* (Pt) are the main root-lesion nematodes causing yield loss in the southern agricultural region of Australia.
- While it needs to be considered, RLN susceptibility does not necessarily preclude chickpea from crop rotations in southern Australia.
- There are consistent varietal differences in Pt resistance within chickpea varieties.
- Successful management relies on:
  - farm hygiene to keep fields free of RLN
  - soil testing to determine whether RLN are an issue and which species are present
  - growing tolerant varieties when RLN are present, to maximise yields
  - rotating with resistant crops to keep RLN at low levels.
- Nitrogen fertilisers, particularly those containing ammonium, have been found to limit nematode infection. RLN may adversely affect the growth and yield of the chickpeas in some cases, but has more effect on the following cereal crop. [1]

Nematodes are microscopic worms that are sometimes known as 'roundworms' or 'eelworms'. Those living in soil are generally small (less than 1 mm long and only 15–20 µm wide) and can only be seen with a microscope. Nematodes are common soil pests that feed on the roots of a wide range of crop plants in all agricultural areas of southern Australia, irrespective of soil type and rainfall. Nematodes multiply on susceptible hosts. Consequently, as nematode populations increase, crop production is limited. Damaged roots have less efficient water and nutrient uptake, and plants are also less able to tolerate other stresses such as drought. [2]

VIDEOS

1. GCTV6: Root-lesion nematodes.

8.1 Root-Lesion nematode (RLN)

Chickpea growers in southern Australia need to be aware of RLN and its impacts. It should not necessarily deter them from growing chickpea in their crop rotations. RLN can however be a major issue in northern Australian crops and rotations.

Key points:

- RLNs are species of *Pratylenchus* nematodes that feed on the roots of crops and can cause yield loss.
- The main RLN species causing damage in the Southern region are *Pratylenchus* neglectus and *P. thornei*.
- RLN reduce development of lateral roots, which decreases the ability of plants to extract water and nutrients.
- The *Pratylenchus* species present in the soil will affect choice of management practices, in particular rotations.
- The host range of RLN is broad and includes cereals, oilseeds, grain legumes and pastures, as well as many broadleaf and grass weeds. [3]

1 Pulse Australia (2007) Chickpeas in South Australia and Victoria. http://www.pulseaus.com.au/storage/app/media/crops/2007_Chickpeas-SA-Vic.pdf

2 S Collins (2016) Root lesion and burrowing nematodes: diagnosis and management. DAFWA, https://www.agric.wa.gov.au/barley/root-lesion-and-burrowing-nematodes-diagnosis-and-management

3 GRDC Tips and Tactics Root-lesion nematodes – Western Region (2015), https://grdc.com.au/Resources/Factsheets/2015/03/Root-Lesion-Nematodes

- Crop rotation and resistant cultivar selection are the keys to management of RLN. Growers need to know which species of RLN are present as cultivars resistant to one nematode species may be susceptible to another, so suitable rotations will vary.
- Become familiar with root and crop symptoms associated with nematode damage.
- Make use of available testing services like PreDicta B™ to determine nematode species and levels.
- Consider the influence of soil nematode levels not only on the current, but also on subsequent, crops in the rotation. [4]

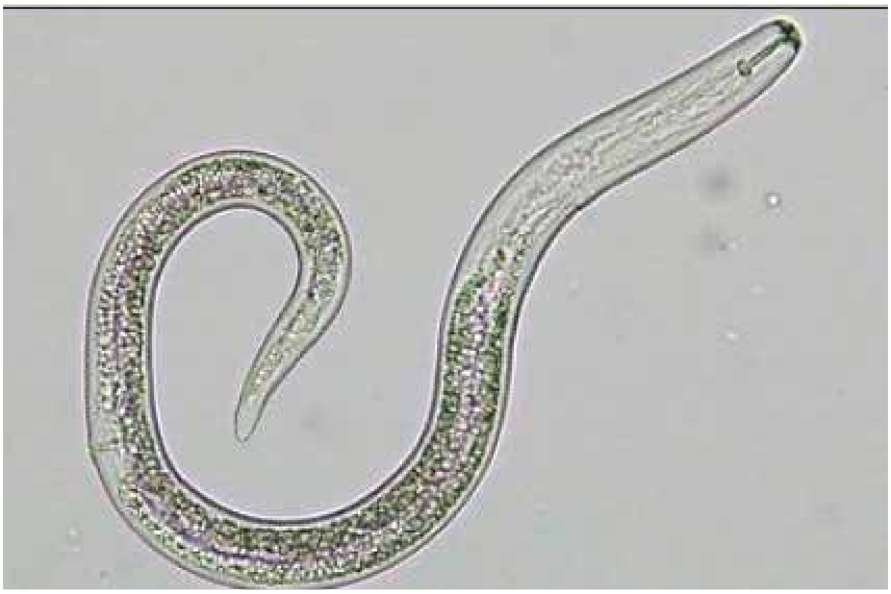

Photo 1: *Microscope image of a root-lesion nematode. Notice the syringe-like 'stylet' at the head end, which is used for extracting nutrients from the plant root. This nematode is less than 1 mm long.*

Photo by Sean Kelly, DAFWA, Nematology

RLN are microscopic migratory endoparasites (Photo 1). This means that RLN enter roots, feed on cell contents then either remain to continue feeding within the same root or exit and move to nearby root systems. This process damages the root system making water and nutrient uptake less efficient, therefore plants are less able to tolerate other stresses. Currently, RLN damage is estimated to cause crop losses in the order of $190 million p.a. in Southern and Western Australia (Figures 1 and 2). [5]

4 S Collins, S Kelly, H Hunter, B MacLeod, L Debrincat, J Teasdale, C Versteeg, X Zhang (2013) Pratylenchus teres—WA's home grown root-lesion nematode (RLN) and its unique impacts on broadacre crops. DAFWA 12 March 2013, https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/03/Pratylenchus-teres-WAs-home-grown-Root-Lesion-Nematode

5 Vanstone *et al.* (2008) Managing nematode pests in the southern and western regions of the Australian cereal industry: continuing progress in a challenging environment. Australasian Plant Pathology Society, Australasian Plant Pathology 37(3), 220–234.

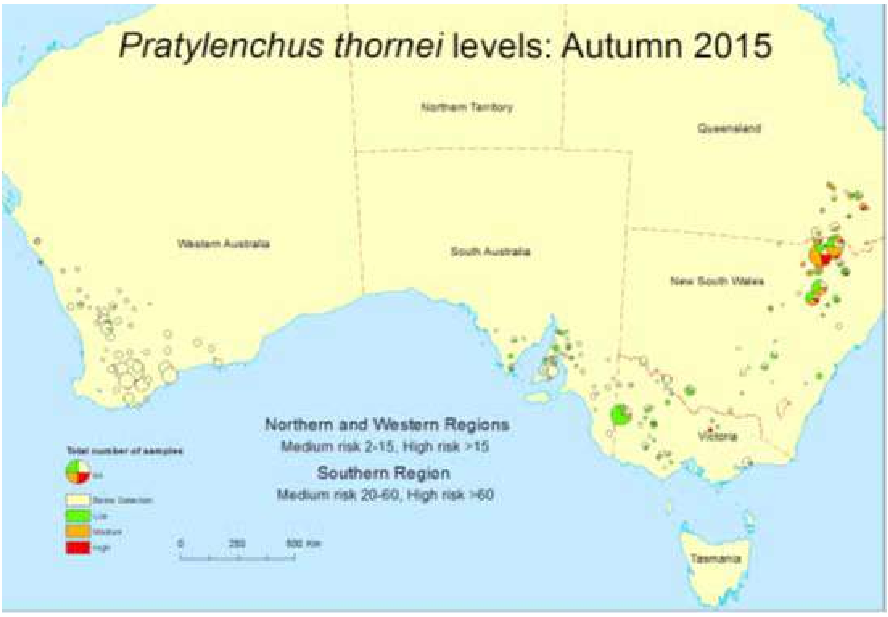

Figure 1: *Pratylenchus thornei levels in Autumn 2015.*

Source: GRDC

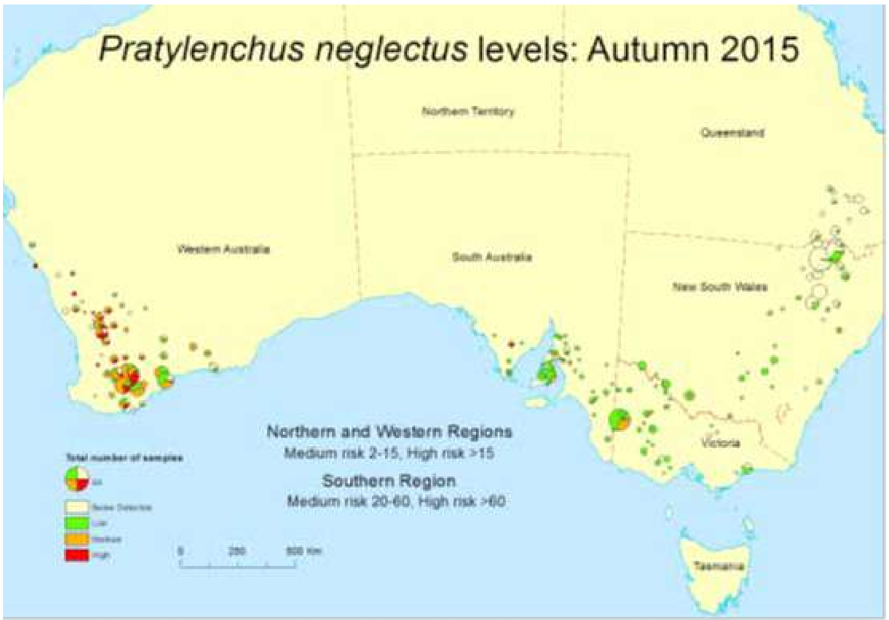

Figure 2: *Pratylenchus neglectus levels in Autumn 2015.*

Source: GRDC

Surveys also found that RLN is at yield-limiting levels in at least 40% of paddocks (Table 1). Several types of RLN are responsible and paddocks usually have one or more species. It is imperative that in field diagnoses, the species of RLN is correctly identified to enable growers to deploy appropriate crop cultivars and species to minimise current and future losses.

Pratylenchus penetrans is rare in broadacre crops but can cause severe damage to some crops. In the Southern Region, high densities of RLN generally cause yield losses of 10–20% in wheat crops. In field in Victoria and South Australian (2011 to

2013), P. thornei reduced grain yield in intolerant varieties by 2–12%, and P. neglectus reduced grain yield by 2–8%. [6]

Table 1: *Grain yield loss (%) caused by root-lesion nematodes in Victoria and South Australia. Values are average percentage yield loss in the five most intolerant wheat varieties.*

	P. thornei		*P. neglectus*	
	South Australia	Victoria	South Australia	Victoria
2011	7.7	12.2	No trial	4.3
2012	9.0	5.3	8.0	6.6
2013	No trial	2.4	3.8	2.6

Source: GRDC Tips and Tactics Root-lesion nematodes—Southern Region (2015), GRDC

IN FOCUS

Effect of field crops on population densities of RLNs in South-eastern Australia; Part 1 and 2.

Eighty-one cultivars from 12 field crop species were assessed for suitability as hosts to the RLN, *Pratylenchus neglectus*, in two field trials. Host status was assessed on the basis of either final *P. neglectus* densities in soil or multiplication rate under different crops. Both techniques gave consistent results for crop and cultivar ranking, and it was therefore concluded that, in these trials, final population density could be used for screening cultivars for resistance to *P. neglectus*. Differences were observed among crops and cultivars for host suitability to *P. neglectus*. Chickpea, wheat, and canola were good hosts, while barley, oat, durum wheat, medic, and vetch were moderate hosts. Field pea, faba bean, and triticale were poor hosts. A range in host suitability was observed for wheat, barley, and oat cultivars. [7]

VIDEOS

2. Root-lesion nematodes. Resistant cereal varieties have surprising impact on RLN numbers.

8.1.1 Varietal resistance or tolerance

A tolerant crop yields well when large populations of RLN are present (in contrast to an intolerant crop). A resistant crop does not allow RLN to reproduce and increase in number (in contrast to a susceptible crop).

Chickpeas are susceptible to *P. neglectus*, *P. thornei* and *P. penetrans*. [8] Chickpea varieties differ in their resistance and tolerance to RLN but are generally considered more susceptible (allowing nematodes to multiply) than field pea, faba bean and lupin, but less so than wheat. While older chickpea varieties were a host for the RLN *(Pratylenchus neglectus, P. thornei)*, newer varieties are not as susceptible to RLN multiplication. [9]

Research in the Northern growing region indicates that there are consistent differences in *Pt* resistance between commercial chickpea varieties.

6 S Collins, S Kelly, H Hunter, B MacLeod, L Debrincat, J Teasdale, C Versteeg, X Zhang (2013) Pratylenchus teres—WA's home grown root-lesion nematode (RLN) and its unique impacts on broadacre crops. GRDC Update Papers 12 March 2013, https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/03/Pratylenchus-teres-WAs-home-grown-Root-Lesion-Nematode

7 SP Taylor, G J Hollaway, CH Hunt (2000) Effect of field crops on population densities of *Pratylenchus neglectus* and *P. thornei* in south-eastern Australia; Part 1: *P. neglectus*. Journal of Nematology, 32(4S): 591–599, https://www.ncbi.nlm.nih.gov/pmc/articles/PMC2620495/

8 GRDC Tips and Tactics Root-lesion nematodes—Western Region (2015), https://grdc.com.au/Resources/Factsheets/2015/03/Root-Lesion-Nematodes

9 Pulse Australia (2015) Chickpea production: Southern and Western region. Pulse Australia November 2015, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/southern-guide

8.1.2 Damage caused by RLN

RLNs cause poor plant growth in situations that otherwise appear favourable. They attack cereals and pulses and are thus a threat to the whole farming system. The nematodes feed and multiply on and in the roots of chickpea plants and, when in sufficient numbers, reduce growth and yield. RLN numbers build up steadily under susceptible crops and cause decreasing yields over several years. Intolerant chickpea varieties can lose up to 20% yield when nematode populations are high. [10] Yield losses in the southern region are variable and currently under investigation, but present estimates for intolerant varieties indicate a 1% yield loss per 2 nematodes per gram soil. [11]

8.1.3 Symptoms

RLNs are microscopic and cannot be seen with the naked eye in the soil or in plants. Aboveground symptoms are often indistinct and difficult to identify. The first signs are poor establishment, stunting and plants possibly wilting despite moist soil. Nematodes are usually distributed unevenly across a paddock, resulting in irregular crop growth (Photo 2). Sometimes symptoms are confused with nutrient deficiency and they can be exacerbated by a lack of nutrients. The typical symptom in the southern region is large areas of poor crop vigour due to poor root growth.

Photo 2: *Chickpea field infested with nematode.*

Source: IIPR

When roots are damaged by RLN, the plants become less efficient at taking up water and nutrients, and less able to tolerate stresses such as drought or nutrient deficiencies. Depending on the extent of damage and the growing conditions, affected plants may partly recover if the rate of new root growth exceeds the rate at which nematodes damage the roots.

Chickpea roots can show distinct dark-brown–orange lesions at early stages of infection and the lateral roots can be severely stunted and reduced in number. The root cortex (or outer root layer) is damaged and it may disintegrate.

Diagnosis is difficult and can be confirmed only with laboratory testing, particularly to identify the species because all RLN species cause identical symptoms. The PreDicta

10 DPI QLD (2009) Root-Lesion nematode management. https://www.daf.qld.gov.au/__data/assets/pdf_file/0010/58870/Root-Lesion-Nematode-Brochure.pdf

11 A McKay Root-lesion nematode—South Australia. Soilquality Fact Sheet,Soilquality.org.au, http://www.soilquality.org.au/factsheets/root-lesion-nematode-south-australia

VIDEOS

3. How to diagnose root-lesion nematode.

B™ soil test (SARDI Diagnostic Services) is a useful tool for several nematode species and is available through accredited agronomists. [12]

RLNs are microscopic and cannot be seen with the naked eye in the soil or in plants. The most reliable way to confirm the presence of RLNs is to test your farm soil. Nematodes are extracted from the soil for identification and to determine their population size. Look out for tell-tale signs of nematode infection in the roots and symptoms in the plant tops and if seen submit soil and root samples for nematode assessment. [13]

Root damage—dark lesions and poor root structure

RLNs invade the root tissue resulting in light browning of the roots or localised deep brown lesions (Photo 3). However, these lesions can be difficult to see on roots. The damage to the roots and the appearance of the lesions can be made worse by fungi and bacteria also entering the wounded roots. Roots infected by RLNs are poorly branched, lack root hairs and do not grow deeply into the soil profile. Such root systems are inefficient in taking up soil nutrients (particularly nitrogen, phosphorus and zinc under Northern region conditions) and soil water.

Photo 3: *Brown lesions indicate entry points of RLN on chickpea roots.*

Photo: Vivien Vanstone, DAFWA, Nematology

Plant tops—stunted, yellow lower leaves, wilting

When RLNs are present in very high numbers the lower leaves of wheat plants are yellow and the plants are stunted with reduced tillering. There is poor canopy closure so that the crop rows appear more open. The tops of the plants may exhibit symptoms of nutrient deficiency (nitrogen, phosphorus and zinc) when the roots are damaged by RLNs. Infected crops can wilt prematurely, particularly when conditions become dry later in the season because the damaged root systems are inefficient at

12 GRDC Tips and Tactics Root-lesion nematodes—Western region (2015), https://grdc.com.au/Resources/Factsheets/2015/03/Root-Lesion-Nematodes

13 A McKay, Root-lesion nematode—South Australia. Soilquality Fact Sheet,Soilquality.org.au, http://www.soilquality.org.au/factsheets/root-lesion-nematode-south-australia

taking up stored soil moisture. With good seasonal rainfall, wilting is less evident and plants may appear nitrogen deficient. [14]

8.1.4 Conditions favouring development

The adult RLNs are nearly all self-fertile females. They lay eggs inside roots and pass through a complete life cycle in about six weeks under favourable conditions (warm, moist soil) and so pass through several generations in the life of one host crop (Figure 3). The nematodes survive through fallow periods, particularly in the subsoil where they escape the hot, drying conditions of the surface soil. In drought or as plants and soil dry out in late spring, the nematodes can dehydrate (anhydrobiosis) to further aid their survival until favourable conditions return. [15]

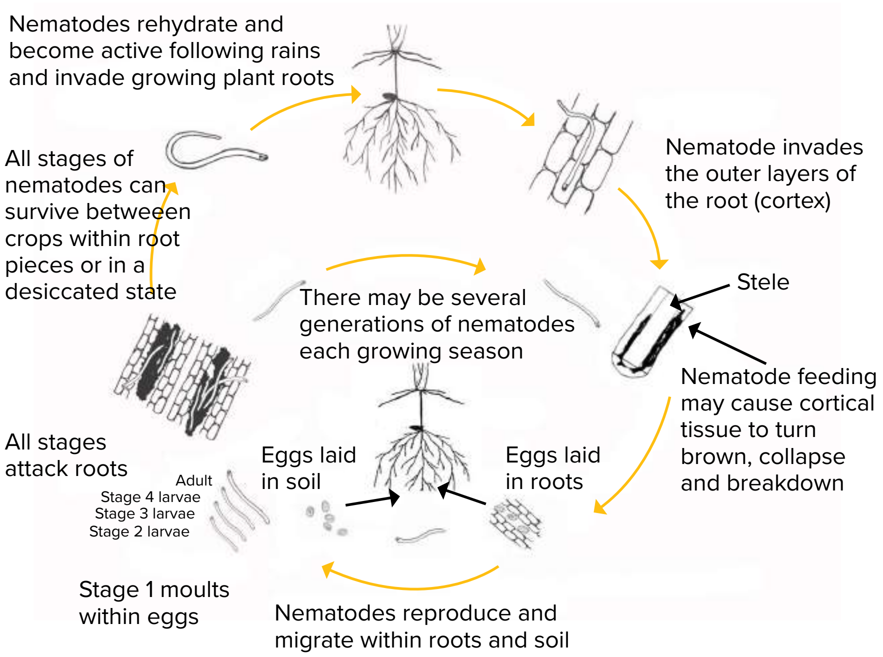

Figure 3: *Disease cycle and damage of root-lesion nematode, adapted from: GN Agrios (1997).*

Illustration: Kylie Fowler, GRDC

8.1.5 Thresholds for control

Yield losses in the southern region are variable and currently under investigation, but present estimates for intolerant varieties indicate a 1% yield loss per 2 nematodes per gram soil. [16]

In the Southern region, *Pratylenchus thornei* at 10 nematodes/g soil can cause grain yield losses of 10–15% in the intolerant wheat variety DERRIMUT, depending on seasonal conditions. [17]

14 DPI QLD (2009) Root-Lesion nematode management. https://www.daf.qld.gov.au/__data/assets/pdf_file/0010/58870/Root-Lesion-Nematode-Brochure.pdf

15 DPI QLD (2009) Root-Lesion nematode management. https://www.daf.qld.gov.au/__data/assets/pdf_file/0010/58870/Root-Lesion-Nematode-Brochure.pdf

16 A McKay, Root-lesion nematode—South Australia. Soilquality Fact Sheet, Soilquality.org.au, http://www.soilquality.org.au/factsheets/root-lesion-nematode-south-australia

17 GRDC Tips and Tactics Root-lesion nematodes—Southern region (2015), https://grdc.com.au/resources-and-publications/all-publications/factsheets/2015/03/tt-rootlesionnematodes

8.1.6 Management of RLN

There are four key strategies for the management of RLN:

1. Test soil for nematodes in a laboratory.
2. Protect paddocks that are free of nematodes by controlling soil and water run-off and cleaning machinery; plant nematode-free paddocks first.
3. Choose tolerant varieties to maximise yields. Tolerant varieties grow and yield well when RLN are present. [18]
4. Rotate with resistant crops to prevent increases in RLN. When large populations of RLN are detected, you may need to grow at least two resistant crops consecutively to decrease populations. In addition, ensure that fertiliser is applied at the recommended rate so that the yield potential of tolerant varieties is achieved.

Figure 4 is a simplified chart that highlights the critical first step in the management of RLN is to test your soil and determine whether or not you have an issue to manage.

VIDEOS

4. What lies beneath – managing RLNs to combat yield loss.

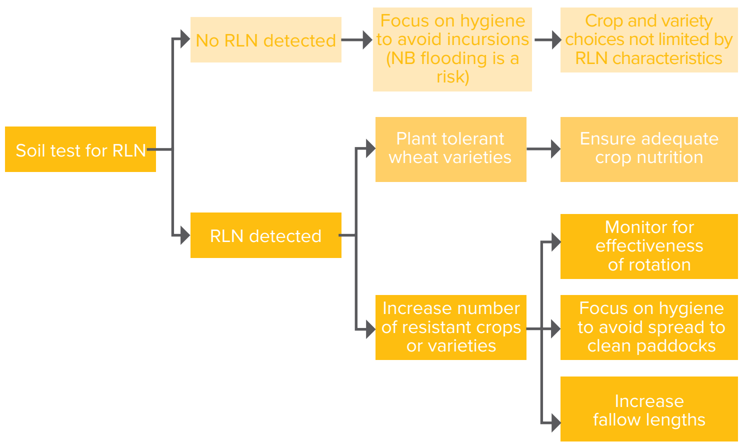

Figure 4: *RLN management flow chart.*

Source: GRDC

Monitoring

Observation and monitoring of above and below ground symptoms of plant disease, followed by diagnosis of the cause(s) of any root disease, is the first step in implementing effective management. Although little can be done during the current cropping season to ameliorate nematode symptoms, the information will be crucial in planning effective rotations of crop species and varieties in following seasons.

Commercial pre-season testing of soil by the PreDicta B™ root disease testing service determines levels of *P. neglectus* and *P. thornei* present using a DNA detection technique. Currently, this test is limited in its ability to detect levels of *P. penetrans* in the soil, and any results from southern Australian soils using PreDicta B™ should be confirmed by traditional laboratory extraction and microscopic examination. During the season, plants with suspected RLN infections should be sent to a laboratory for extraction and identification. [19]

If RLN infestation is suspected, growers are advised to check the crop roots. Carefully digging up and washing the soil from the roots of an infected plant can reveal evidence of infestation in the roots, which warrants laboratory analysis.

18 KJ Owen, J Sheedy, N Seymour (2013) Root lesion nematode in Queensland. Soilquality Fact Sheet, Soilquality.org.au, http://soilquality.org.au/factsheets/root-lesion-nematode-in-queensland

19 A McKay, Root-lesion nematode—South Australia. Soilquality.com.au Fact Sheet, Soilquality.org.au, http://www.soilquality.org.au/factsheets/root-lesion-nematode-south-australia

On-farm

Growers are advised to check the roots of the host crops if they suspect RLN infestations. Carefully dig up roots, then wash the soil from the roots of an infected plant and inspect for symptoms (as above). If evidence of infestation in the roots is observed, then a laboratory analysis or a PreDicta B™ test can be used to determine species and density.

Commercial

A DNA test, PreDicta B™, is commercially available around Australia and growers should contact their state department of agriculture for advice.

Grain producers can access PreDicta B™ via agronomists accredited by SARDI to interpret the results and provide advice on management options to reduce the risk of yield loss.

PreDicta B™ samples are processed weekly from February to mid-May (prior to crops being sown) to assist with planning the cropping program. Crop diagnosis is best achieved by sending samples of affected plants to your local plant pathology laboratory.

Postal Address for PreDicta B™ samples: C/- SARDI RDTS, Locked Bag 100 Glen Osmond SA 5064

Courier address: SARDI Molecular Diagnostics Group Plant Research Centre, Gate 2B Hartley Grove, URRBRAE SA 5064. [20]

Soil testing

Vertical distribution of *P. thornei* in soil is variable. Some paddocks have relatively uniform populations down to 30 cm or even 60 cm, some will have highest *P. thornei* counts at 0–15 cm depth, whereas other paddocks will have *P. thornei* populations increasing at greater depths, e.g. 30–60 cm. Although detailed knowledge of the distribution may be helpful, the majority of on-farm management decisions will be based on presence or absence of *P. thornei* confirmed by sampling. [21]

When to collect samples

The best time for sampling varies between crops, and is related to the growth stage of the crop and the objective of sampling. Many species of nematodes increase to high levels during the growing season and reduce to low numbers during the dry season. This is more easily seen in annual crops than in perennial and tree crops.

Sampling equipment:

- clean bucket for collecting samples
- soil probe (Photo 4) or shovel/spade
- plastic bags to hold 500 g of soil
- labels
- waterproof marker

20 GRDC Tips and Tactics Root-lesion nematodes—Southern region (2015), https://grdc.com.au/resources-and-publications/all-publications/factsheets/2015/03/tt-rootlesionnematodes

21 KJ Owen, J Sheedy, N Seymour (2013) Root-lesion nematode in Queensland. Soilquality Fact Sheet,Soilquality.org.au, http://www.soilquality.org.au/factsheets/root-lesion-nematode-in-queensland

Photo 4: *Soil sampling probe.*

Source: United soybean board

How to sample

Fallow or bare fields:

Do not collect samples when field is dry or extremely wet. For sampling, the field should be divided into 1–2 hectare blocks. Take about 20–30 cores/sub-samples of soil, at 15–20 cm depth from an area of 1–2 hectares. Collect these sub-samples at every 10–20 metres in a 'W' or in a 'Zigzag' pattern (Figure 5). Place sub-samples in a bucket and mix thoroughly with hands, and collect a 500 g composite sample in a labelled plastic bag.

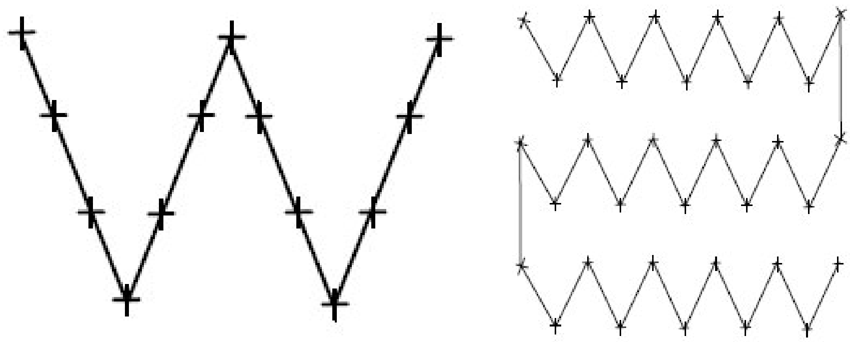

Figure 5: *'W' pattern (left) and 'Zigzag' pattern (right) for randomising soil sampling in a paddock.*

Source: AgVic

Field crops:

Nematodes do not necessarily occur uniformly throughout a field, therefore several sub-samples must be taken from across the field, and then combined. Collect 20–30 random sub-samples from each block of 1–2 hectares. Samples should be taken directly from the root zone. Mix sub-samples thoroughly and place 500 g of soil, with roots, in a plastic bag, for laboratory analysis. Because nematode damage within a crop can be patchy, collect samples from healthy plants, as well as from plants showing symptoms of decline. Keep these samples separate and label them as 'good' and 'bad' samples.

Care of samples

Place all samples in plastic bags to prevent drying. Generally, plant-parasitic nematodes remain alive at temperature between 5°C and 40°C, and die within seconds when exposed to temperatures above 50°C. Keep samples in a cool place at all times. Do not refrigerate samples. Do not leave samples exposed in the field, or in a vehicle, on very hot days. Do not wrap roots or any other plant material in damp tissue. Leave roots with soil in bag. Place other plant material in a separate plastic bag.

Label and information

Label samples with identification numbers and provide the following information on a separate sheet of paper:

- name and address of the grower as well as sender
- crop plant, symptoms and estimated damage
- a sketch map of the diseased area and the sampling site, and also an indication of the topography of the field
- cropping history of the field
- fertilisers, pesticides and herbicides applied
- relevant weather conditions and watering or drainage conditions

It is necessary to provide the above information so the results of the analysis can be interpreted correctly and satisfactorily. [22]

Control strategies

- Well-managed rotations with resistant or non-host break-crops are vital. To limit RLN populations, avoid consecutive host crops.

Use a state department of agriculture Crop Variety Guide to choose varieties with high resistance ratings, which result in fewer nematodes remaining in the soil to infect subsequent crops.

- Reducing RLN can lead to higher yields in following cereal crops.
- Healthy soils and good nutrition can partly alleviate RLN damage through good crop establishment, and healthier plants may recover more readily from infestation under more suitable growing conditions.
- Observe crop roots to monitor development of symptoms.
- Weeds can host parasitic nematodes within and between cropping sequences, so choice of pasture species and control of host weed species and crop volunteers is important. [23]

Nematicides

Nematicides are not used commercially in broadacre cropping in Australia. They are not recommended because of their cost and mammalian toxicity, and because rotational crops are available for nematode management. If they were used commercially, their efficacy would likely be poor, particularly in situations where the nematode occurs at depth. Currently, no nematicides are registered for use on broadacre crops in Australia. [24]

Variety choice and crop rotation options

Varietal choice and crop rotation are currently our most effective management tools for RLN. However, the focus is on two different characteristics: tolerance, i.e. ability of the variety to yield under RLN pressure; and resistance, i.e. impact of the variety

22 AgVic (2011) Collecting soil and plant samples for nematode analysis. Agriculture Victoria, Ag Note AG1444 July 2011, http://agriculture.vic.gov.au/agriculture/pests-diseases-and-weeds/plant-diseases/fruit-and-nuts/stone-fruit-diseases/collecting-soil-and-plant-samples-for-nematode-analysis

23 GRDC (2010) Plant parasitic nematodes fact sheet Southern and Western region. 28 October 2010, https://grdc.com.au/Resources/Bookshop/2010/10/Plant-Parasitic-Nematodes-Fact-Sheet-Southern-Western-Region

24 GRDC Tips and Tactics Root-lesion nematodes—Western region (2015), https://grdc.com.au/Resources/Factsheets/2015/03/Root-Lesion-Nematodes

on RLN build-up. Note that varieties and crops often have varied tolerance and resistance levels to *P. thornei* and P. neglectus.

Chickpea grown in rotations with wheat (*Triticum aestivum*) can reduce the build-up of pathogens of cereals such as *Fusarium pseudograminearum* (responsible for crown rot), improve soil nitrogen (N) fertility, and facilitate control of grass weeds. However, offsetting these benefits, populations of RLN increase with chickpea rotations, reducing its yield and negatively affecting the yield of subsequent intolerant wheat and other crops. [25]

Summer crops (where grown) have an important role in management of RLN. Research shows that when *P. thornei* is present in high numbers, two or more resistant crops in sequence are needed to reduce populations to low enough levels to avoid yield loss in the following intolerant, susceptible wheat crops. Summer crops that are partially resistant or poor hosts of *P. neglectus* include sunflower, mungbean, soybean and cowpea. When these crops are grown, populations of *P. neglectus* do not increase because the crops do not allow the nematode to reproduce. [26]

MORE INFORMATION

For more information, see the GRDC Tips and Tactics Root-Lesion Nematode Factsheet – Southern Region.

IN FOCUS

Yield response in chickpea cultivars and wheat following crop rotations affecting population densities of *Pratylenchus thornei* and arbuscular mycorrhizal fungi.

In Australia, RLN significantly reduces chickpea and wheat yields. Yield losses from RLN have been determined through use of nematicide; however, nematicide does not control nematodes in Vertosol subsoils in Australia. The alternative strategy of assessing yield response, by using crop rotation with resistant and susceptible crops to manipulate nematode populations, is poorly documented for chickpea. One study tested the effectiveness of crop rotation and nematicide against *P. thornei* populations for assessing yield loss in chickpea in the Northern cropping region.

Canola, linseed and fallow treatments reduced *P. thornei* populations, but low mycorrhizal spore levels in the soil after canola and fallow treatments were associated with low chickpea yield. Mycorrhizal spore densities are also imortant in maximising the effectiveness of crop rotations.

Canaryseed kept *P. thornei* populations low throughout the soil profile and maintained mycorrhizal spore densities, resulting in grain yield increases of up to 25% for chickpea cultivars and 55% for wheat when pre-cropped with canaryseed compared with wheat. Tolerance indices for chickpeas based on yield differences after paired wheat and canaryseed plots ranged from 80% for cv. Tyson to 95% for cv. Lasseter and this strategy is recommended for future use in assessing tolerance. [27]

25 RA Reen, JP Thompson, TG Clewett, JG Sheedy, KL Bell (2014) Yield response in chickpea cultivars and wheat following crop rotations affecting population densities of *Pratylenchus thornei* and arbuscular mycorrhizal fungi. Crop and Pasture Science, 65(5), 428-441.

26 K Owen, T Clewett, J Thompson (2013) Summer crop decisions and root-lesion nematodes: crop rotations to manage nematodes—key decision points for the latter half of the year, Bellata. GRDC Update Papers, 16 July 2013, https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/07/Summer-crop-decisions-and-root-lesion-nematodes

27 RA Reen, JP Thompson, TG Clewett, JG Sheedy, KL Bell (2014). Yield response in chickpea cultivars and wheat following crop rotations affecting population densities of *Pratylenchus thornei* and arbuscular mycorrhizal fungi. Crop and Pasture Science, 65(5), 428-441.

Fallow

Populations of RLN will decrease during a 'clean' fallow, but the process is slow and expensive in lost 'potential' income. [28]

8.1.7 Breeding resistance

IN FOCUS

Hybridisation of Australian chickpea cultivars with wild *Cicer* spp. increases resistance to root-lesion nematodes (*Pratylenchus thornei* and *P. neglectus*)

Development of commercial cultivars from wild hybrid parents with the high levels of resistance to *P. thornei* and *P. neglectus* will be most valuable for areas of the Australian grain region and other parts of the world where alternating chickpea and wheat crops are the preferred rotation.

Australian and international chickpea (*Cicer arietinum*) cultivars and germplasm accessions, and wild annual *Cicer* spp. in the primary and secondary gene pools, were assessed in glasshouse experiments for levels of resistance to the root-lesion nematodes *Pratylenchus thornei* and *P. neglectus*.

Lines were grown in replicated experiments in pasteurised soil inoculated with a pure culture of either *P. thornei* or *P. neglectus* and the population density of the nematodes in the soil plus roots after 16 weeks' growth was used as a measure of resistance. Combined statistical analyses of experiments (nine for *P. thornei* and four for *P. neglectus*) were conducted and genotypes were assessed using best linear unbiased predictions. Australian and international chickpea cultivars possessed a similar range of susceptibilities through to partial resistance. Wild relatives from both the primary (*C. reticulatum* and *C. echinospermum*) and secondary (*C. bijugum*) gene pools of chickpea were generally more resistant than commercial chickpea cultivars to either *P. thornei* or *P. neglectus* or both. Wild relatives of chickpea have probably evolved to have resistance to endemic root-lesion nematodes whereas modern chickpea cultivars constitute a narrower gene pool with respect to nematode resistance. Resistant accessions of *C. reticulatum* and *C. echinospermum* were crossed and topcrossed with desi chickpea cultivars and resistant F4 lines were obtained. [29]

28 R Daniel (2013) Managing root-lesion nematodes: how important are crop and variety choice? GRDC Update Paper 16 July 2013, https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/07/Managing-root-lesion-nematodes-how-important-are-crop-and-variety-choice

29 JP Thompson, RA Reen, TG Clewett, JG Sheedy, AM Kelly, BJ Gogel, EJ Knights (2011). Hybridisation of Australian chickpea cultivars with wild *Cicer* spp. increases resistance to root-lesion nematodes (*Pratylenchus thornei and P. neglectus*). Australasian Plant Pathology Society, Australasian Plant Pathology, 40(6), 601–611.

IN FOCUS

Highly heritable resistance to root-lesion nematode (*Pratylenchus thornei*) in Australian chickpea germplasm observed using an optimised glasshouse method and multi-environment trial analysis

Pratylenchus thornei is a RLN of economic significance in the grain growing regions of Australia. Chickpea (*Cicer arietinum*) is a significant legume crop grown throughout these regions, but previous testing found most cultivars were susceptible to *P. thornei*. Therefore, improved resistance to *P. thornei* is an important objective of the Australian chickpea breeding program. Results demonstrate that resistance to *P. thornei* in chickpea is highly heritable and can be effectively selected in a limited set of environments. The improved resistance found in a number of the newer chickpea cultivars tested shows that some advances have been made in the *P. thornei* resistance of Australian chickpea cultivars, and that further targeted breeding and selection should provide incremental improvements. [30]

VIDEOS

5. GCTV9: Crown rot and root-lesion nematodes.

MORE INFORMATION

The additive yield impact of root-lesion nematode and crown rot

8.2 Nematodes and crown rot

There is increasing evidence for the enhancing effect of nematodes on levels of crown rot (CR) in cereals. After an extensive NSW farm survey exploring the effect of CR on crops, the researchers concluded that where *P. thornei* (Pt) combines with high levels of CR (a common scenario), yield losses can be exacerbated if varieties are susceptible to Pt. Instead of a 10% yield loss from Pt in a susceptible variety it could be 30–50% if CR is combined with a Pt-intolerant variety. These trials were designed to evaluate the impact of CR on variety yield and quality. However, results strongly suggest that Pt is also having a significant impact on yield performance. The results do not compare the actual levels of yield loss due to the two diseases but indicate there is a greater range in variety Pt tolerance than currently exists for CR tolerance. Put simply, variety choice appears a more valuable tool when under Pt pressure than as a tool for CR management. [31]

30 MS Rodda, KB Hobson, CR Forknall, RP Daniel, JP Fanning, DD Pounsett, JP Thompson (2016). Highly heritable resistance to root-lesion nematode (*Pratylenchus thornei*) in Australian chickpea germplasm observed using an optimised glasshouse method and multi-environment trial analysis. Australasian Plant Pathology Society, Australasian Plant Pathology, 45(3), 309-319.

31 GRDC (2010) The additive yield impact of root lesion nematode and crown rot. GRDC Update Papers 15 September 2010, https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2010/09/THE-ADDITIVE-YIELD-IMPACT-OF-ROOT-LESION-NEMATODE-AND-CROWN-ROT

Diseases

Key messages

- Several foliar fungal diseases, some seedling root diseases, viruses and root-lesion nematode can affect chickpea (Table 1).
- The most significant fungal disease of chickpea is Ascochyta blight. Disease management of chickpea should primarily focus on Ascochyta blight.
- Chickpea crops in southern Australia are being hit by a more virulent strain of the damaging ascochyta blight.
- The diseases Botrytis grey mould, *Botrytis cinerea*, and Sclerotinia white mould (*Sclerotinia sclerotiorum* and *S. minor*) were major diseases of chickpea prior to the incursion of Ascochyta blight, and may again become significant diseases in chickpea varieties resistant to Ascochyta blight. [1]
- Integrated disease management in chickpeas involves paddock selection, variety choice, seed dressing, strategic fungicide use and hygiene.
- Implement an appropriate Ascochyta blight strategy based on rain forecasts and the level of varietal resistance.
- Using a number of integrated disease management (IDM) techniques is more likely to control diseases.
- Stay up to date with local Crop diseases forecasts for your region.

In northern Victoria and southern NSW the important disease constraints to chickpea production are Ascochyta blight (caused by *Ascochyta rabiei*) and grey mould (caused by *Botrytis cinerea*) (Tables 1 and 2).

Unlike northern NSW, Phytophthora root rot is not a widespread production issue and has not been detected in southern chickpea producing areas. These fungal pathogens have the potential to reduce crop yield and seed quality.

Disease management strategies for both diseases have been developed that utilise a range of chemical and non-chemical approaches, such as paddock selection, crop rotation, selection of seed for sowing, variety selection, sowing date and rate, and the strategic use of fungicides (both fungicidal seed dressings and foliar fungicides).

Producers still rely heavily on fungicides and success is dependent on correct disease identification, timing of product application and fungicide choice. These strategies are available from the Pulse Australia website. [2]

Virus management aims at prevention through integrated management practice that involves controlling the virus source, aphid populations and virus transmission into pulse crops.

Rotate legume crops with cereals to reduce virus and vector sources. Where possible avoid close proximity to perennial pastures (eg lucerne) or other crops that host viruses and aphid vectors.

Eliminate summer weeds and self sown pulses that are a ‘green bridge’ as a host for viruses and a refuge for aphids.

1 K Regan (2016) Production packages for kabuli chickpea in Western Australia—post planting guide. DAFWA, https://www.agric.wa.gov.au/chickpeas/production-packages-kabuli-chickpea-western-australia-post-planting-guide

2 K Lindbeck ((2014) Faba bean and chickpea disease management. GRDC Update Papers 31 July 2014, https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/07/Faba-bean-and-chickpea-disease-management

Table 1: *Key features of the main chickpea disorders—at a glance.*

Disorder and cause	Seed-borne?	Symptoms	Distribution and occurrence	Survival and spread	Management
Seed-borne root rot: *Botrytis cinerea* *Ascochyta rabiei*	Yes	Seedlings wilt and die, epicotyl rots	Random individual plants (not patches)	Seed	Quality seed; seed treatment
Phytophthora root rot (PRR): *Phytophthora medicaginis*	No	Rapid wilting and yellowing; defoliation from lower leaves; rotted roots; plants easy to pull up	Patches; poorly drained areas; heavy rainfall; can occur at any time; history of medics, lucerne or PRR	Oospores in soil and residue persist for many years; survives saprophytically; spread by water and soil	Varietal selection; avoid paddocks with history of PRR; rotation; seed treatment
Waterlogging: root anoxia	No	Very rapid death; little defoliation; roots not rotted but may be dark; plants hard to pull up	Patches; poorly drained areas; heavy rainfall; can occur at any time; history of medics, lucerne or PRR	Caused by insufficient supply of oxygen to roots	Avoid low lying or poorly drained paddocks or areas within paddocks
Sclerotinia root and stem rot: *Sclerotinia* spp.	Yes (ad-mixed)	Wilting and death; bleached root, collar and stem tissue; white cottony mould at site of lesion; sclerotia at lesions or inside stems	Root and collar lesions result from direct infection from sclerotia; stem lesions result from airborne ascospores released from sclerotial apothecia, scattered or patches; favoured by denser canopies; wet events	Sclerotia persist in soil for many years; wide host range including pulses, canola, sunflowers and broadleaf weeds but not cereals or grasses	Avoid paddocks with history of sclerotinia of its hosts; rotate with cereals; some varieties more susceptible
Rhizoctonia rot: *Rhizoctonia solani*	?	Death of seedlings, stunting of survivors due to root damage, re-shooting after damping-off of epicotyl	Can be a problem in irrigated crops grown immediately after cotton. Often occurs in 1–5 m stretches of row	Survives as sclerotia and on decomposing trash. Probably present in most soils	Allow time for decomposition of (preceding) crop debris. Tillage should help
Ascochyta blight: *Ascochyta (Phoma) rabiei*	Yes	Ghosting of tissues; lesions with concentric rings of pycnidia; stem stumps; plant death	Small patches enlarge rapidly in wet weather to kill large areas of crop	Chickpea residue very important in spread especially header dust and surface water flow; infected seed; volunteers	Follow chickpea Ascochyta blight management package published annually; includes foliar fungicides
Botrytis grey mould (BGM): *Botrytis cinerea*	Yes/no	Stem, flower pod and leaf lesions covered in grey mould	Occurs later in season when canopy closes and warm humid conditions persist; individual plants or patches	Can flow-on from seed-borne root rot but pathogen has wide host range and airborne spores can blow around; sclerotia can survive in soil	Avoid highly susceptible varieties; plant on wider rows; follow chickpea Ascochyta blight management package
Root-lesion nematodes. *Pratylenchus* spp.	No	General poor growth; small black lesions on lateral roots sometimes visible	Often affects large parts of crop; *P. thornei* more prevalent on high clay soils	Wide host range; survives and spreads in soil; anhydrobiosis allows nematodes to persist for prolonged dry periods	Farm hygiene; rotate with resistant species; grow tolerant varieties

Disorder and cause	Seed-borne?	Symptoms	Distribution and occurrence	Survival and spread	Management
Alfalfa mosaic virus (AMV), Cucumber mosaic virus (CMV)	Yes	Initially bunching, reddening, yellowing, wilting or death of shoot tips; later discolouration	Initially scattered plants often at edges of crop; more common in thin stands	Viruses persist and multiply in weeds and pasture legumes; aphid-borne except for CpCDV (leafhopper)	Establish uniform stand by using recommended sowing rates and times; sowing into standing stubble
Phloem-limited viruses (luteoviruses): BLRV (Bean leaf roll virus), SCRLV (Subterranean clover red leaf virus), BWYV (Beet western yellows virus), SCSV (Subterranean clover stunt virus)	No	Death of entire plant; luteovirus-infected plants often have discoloured phloem	Close to lucerne; seasons or districts with major aphid flights		Cereal stubble deters aphids; grow resistant varieties
CpCDV (Chickpea chlorotic dwarf virus)	?	Reddening, proliferation of axillary branching	Individual or small clusters of plants. Maybe more at edges of crop	? (Leafhopper transmitted)	?
Cucumber mosaic virus (CMV)		Yellowing, stunting, offshoots. The leaves and stems of desi varieties become red/brown. The leaves and stems of kabuli varieties turn yellow	Prevalent in chickpea growing regions. Seasons and districts with major aphid flights	Very wide host range, including most pulses, pastures, horticultural crops and weeds	Virus-free seed-resistant varieties

Source: K Moore, NSW DPI and M Fuhlbohm, Qld Gov.

Table 2: *Key facts about the biology of major chickpea diseases.*

Disease	Survival	Spread	Infection by
Ascochyta blight	Stubble, seed, volunteers	Stubble, seed water-splashed spores	Water-splashed spores
Botrytis grey mould	Stubble, seed, sclerotia, alternative hosts	Stubble, seed, soil, airborne spores	Airborne spores
Phytophthora root rot	Oospores, alternative hosts	Soil and surface water	Waterborne spores
Sclerotinia rot	Sclerotia in soil and seed, alternative hosts	Soil and water, airborne spores	Airborne spores or directly into crowns

Source: Pulse Australia

Table 3: *Disease rating for current chickpea varieties.*

Variety	Botrytis grey mould	Ascochyta blight* (Foliage/Stem)	Ascochyta blight (Pod)	*P. Thornei* (provisional)
Desi				
Howzat$^{\phi}$	MS	S	S	MSp
PBA Slasher$^{\phi}$	S	S	S	MRMS
PBA Striker$^{\phi}$	S	S	S	
Ambar$^{\phi}$	S	MS	S	
Neelam$^{\phi}$	S	MS	S	
PBA Maiden$^{\phi}$	S	S	S	
Kabuli				
Genesis™ 090	S	MS	S	S
Almaz$^{\phi}$	S	MS	S	VS
Genesis™ 079	S	S	S	MR
Genesis™ Kalkee	S	MS	S	MS
PBA Monarch$^{\phi}$	S	S	S	MS

Source: GRDC

MORE INFORMATION

The current and potential costs from diseases of pulse crops in Australia.

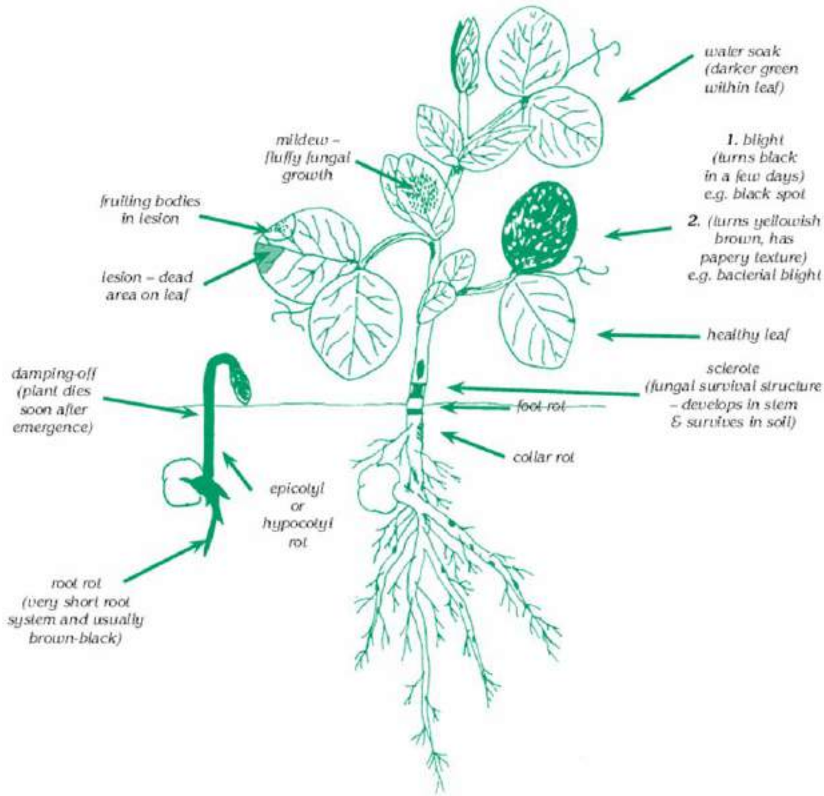

Figure 1: *Pulse disease diagnosis terms.*

Source: Grain Legume Handbook

9.1 Key disease management strategies for chickpeas

- Variety selection is critical. Ideally grow an ascochyta-resistant variety.
- Paddock isolation from chickpea stubble is a high priority (greater than 500 m).
- Paddock history; aim for a break of at least four years between chickpea crops.
- Seed source; use seed from a paddock where disease was not detected.
- Fungicide seed dressing is effective and should be used, especially in high disease risk situations.
- Sowing date; do not sow too early, even with an ascochyta-resistant variety.
- Sowing depth; if using an ascochyta-susceptible variety, sow deeper than normal.
- Sowing rate; aim for 35–50 plants per square metre, depending on the situation and crop type (kabuli or desi).
- Hygiene; reduce disease sources and prevent spread of disease.
- Foliar fungicides; ascochyta-resistant varieties still require foliar fungicide at podding. Success is dependent on monitoring, timeliness of spraying and correct fungicide choice. Early detection and correct disease identification are essential.
- Manage aphids and virus; ground surface cover, healthy plants and crop canopy are important. Control aphids at their source (host) crop.
- Harvest management; harvest early to minimise disease infection of seed.
- Crop desiccation enables even earlier harvest. [3]

9.2 Fungal disease management strategies

Disease management in pulses is critical, and relies on an integrated management approach involving variety choice, crop hygiene and strategic use of fungicides. The initial source of the disease can be from the seed, the soil, the pulse stubble and self-sown seedlings, or in some cases, other plant species. Once the disease is present, the source is then from within the crop itself.

Note that the impact of disease on grain quality in pulses can be far greater than yield loss. This must be accounted for in thresholds because the visual quality of pulses has a huge impact on price for food products. Examples are Ascochyta blight in most pulses and Pea seed-borne mosaic virus in field peas.

A plant disease may be devastating at certain times, and yet under other conditions, it may have little impact. The interactions of host, pathogen and environment are all critical points in disease development, and all can be represented by the disease triangle (Figures 2 and 3). Diseases such as Ascochyta blight and PRR can cause total crop failures very quickly. The effects of BGM and root-lesion nematodes on crop performance and yield usually unfold more slowly, however, they can cause damage quickly when conditions are suitable.

3 W Hawthorne, J Davidson, L McMurray, K Hobson, K Lindbeck, J Brand (2012) Chickpea disease management strategy—Southern region. Pulse Australia, Southern Pulse Bulletin PA 2012 #08, http://pulseaus.com.au/storage/app/media/crops/2012_SPB-Chickpea-disease-management.pdf

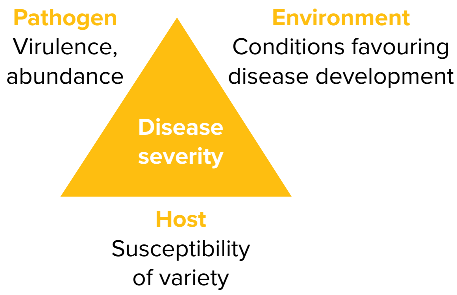

Figure 2: *The virus and some bacterial disease triangle.*

Source: Jones 2012

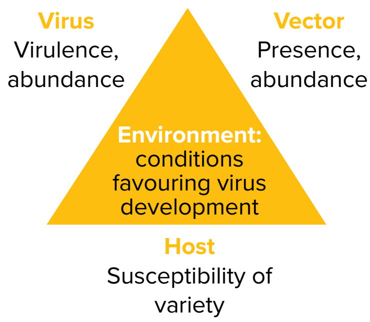

Figure 3: *The virus triangle.*

Source: Agrios 1988

Disease management should be a consideration when planning any rotation, particularly at the beginning of the season. This is especially important for chickpeas where the first defence against diseases begins with paddock selection. Other criteria such as seed quality and treatment are also vitally important. Determine which diseases have the highest priorities to control in the pulse crop being grown, and sow a variety that is resistant to those diseases if possible. Paddock selection and strategic fungicide use are part of the overall program to minimise disease impact. Fungicide disease control strategies alone may not be economic in high-risk situations, particularly if susceptible varieties are grown.

Key strategies:

- **Variety selection**. Growing a resistant variety reduces the need for foliar fungicides.
- **Distance**. Distance from any of last year's stubble of the pulse will affect the amount of infection for some diseases. Aim for a separation of at least 500 m.
- **Paddock history and rotation**. Aim for a break of at least four years between sowing of the same pulse crop. Having a high frequency of crops such as lentil, faba bean, vetch, field pea, chickpea, lathyrus or clover pasture puts pulses at greater risk of diseases such as Phoma blight, Sclerotinia rot and BGM. Ascochyta blight species are more specific to each pulse crop, but 3–4-year rotations are still important. Canola can also increase the risk of Sclerotinia rot.
- **Hygiene**. Take all necessary precautions to prevent the spread of disease. Reduce last year's pulse stubble if erosion is not a risk and remove self-sown pulses before the new crop emerges.

- **Seed source**. Use seed from crops where there were low levels of disease, or preferably no disease, especially at podding. Avoid using seed with known disease infection, particularly with susceptible varieties. Have seed tested for disease status.
- **Fungicide seed dressings**. Dressings are partially effective early in situations of high disease risk, particularly for diseases such as BGM, Phoma blight and Ascochyta blight. They are also effective for seed-borne disease control but not effective on viruses and bacterial diseases.
- **Sowing date**. To minimise foliar disease risk do not sow too early, so avoiding excessive vegetative growth and early canopy closure. Early crop emergence also coincides with greater inoculum pressure from old crop residues nearby. Aim for the optimum sowing window for the pulse and the district.
- **Sowing rate**. Aim for the optimum plant population (depending on region, sowing time, crop type, variety), as denser canopies can lead to greater disease incidence. Adjust seeding rate according to seed size and germination. Avoid double sowing headlands, as the denser crop can be more prone to disease establishment and lodging. Seeding rates below the minimum recommended plant populations will have minimal impact on disease incidence, but reduce potential yield and increase harvest losses.
- **Sowing depth**. Sow deeper than normal any seed lot that is infected with disease to help reduce emergence of infected seedlings. The seeding rate must be adjusted upwards to account for the lower emergence and establishment percentage.
- **Foliar fungicide applications**. Disease-resistant varieties do not require the same regular foliar fungicide program that susceptible varieties need to control foliar diseases. Some pulses may require fungicide treatment for BGM if a dense canopy exists. Successful disease control with fungicides depends on timeliness of spraying, the weather conditions that follow, and the susceptibility of the variety grown. Monitoring for early detection and correct disease identification is essential. Correct fungicide choice is also critical.
- **Controlling aphids**. This may reduce the spread of viruses, but not eliminate them. Strategic or regular insecticide treatments are unlikely to be successful or economic. A virus can be spread by aphids before the aphids are detected.
- **Harvest management**. Early harvest will help to reduce disease infection of seed, and is also important for grain quality and to minimise harvest losses. Crop desiccation enables even earlier harvest. Moisture contents of up to 14% are allowable at delivery. Do not prematurely desiccate as this can affect grain quality. [4]

9.3 Integrated Disease Management

Disease management in chickpeas is critical and relies heavily on an integrated management package involving paddock selection, variety choice, strategic fungicide use and crop hygiene.

The appropriate Ascochyta blight control strategy is then adopted by determining the level of risk in combination with climatic conditions and the level of resistance afforded by the variety chosen.

Disease control strategies may not be economic in high-risk situations if varieties susceptible to Ascochyta blight are grown. [5]

IDM (Integrated Disease Management) is an integrated approach of crop management to reduce chemical inputs and resolve ecological problems. Although originally developed for insect pest management, IPM programs now encompass diseases, weeds, and other pests.

4 Pulse Australia (2013) Southern chickpea best management practices training course manual—2013. Pulse Australia Limited.

5 Pulse Australia (2011) Chickpea Integrated Disease Management. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/idm-strategies

IDM is performed in three stages: prevention, observation and intervention. It is aimed at significantly reducing or eliminating use of pesticides while managing pest populations at an acceptable level.

An IDM system is designed around six basic components:

1. Acceptable disease levels:

- Emphasis is on economical control, not eradication.
- Elimination of the disease is often impossible, and can be economically expensive, environmentally unsafe, and frequently not achievable. IDM programs work to establish acceptable disease levels (action thresholds) and then apply controls if those thresholds are about to be exceeded. Thresholds are specific for disease and site. What is acceptable at one site may not be acceptable at another site or for another crop. Allowing some disease to be present at a reasonable threshold means that selection pressure for resistance pathogens is reduced.

2. Preventive cultural practices:

- Use varieties best suited to local growing conditions and with adequate disease resistance.
- Maintaining healthy crops is the first line of defence, together with plant hygiene and crop sanitation. Crop canopy management is also very important in pulses; hence, time of sowing, row spacing and plant density and variety attributes become important.

3. Monitoring:

- Regular observation is the key to IDM.
- Observation is broken into inspection and then identification. Visual inspection, spore traps, and other measuring tools are used to monitor disease levels. Accurate disease identification is critical to a successful program. Record keeping is essential, as is a thorough knowledge of the behaviour and reproductive cycles of target pests.
- Diseases are dependent on specific temperature and moisture regimes to develop (e.g. rust requires warm temperatures, Ascochyta blight often requires colder temperatures). Monitor the climatic conditions and rain likelihood to determine when a specific disease outbreak is likely.

4. Mechanical controls:

- Should a disease reach unacceptable levels, mechanical methods may be needed for crop hygiene (e.g. burning or ploughing in pulse stubble, removing hay, cultivating self-sown seedlings).

5. Biological controls:

- Crop rotation and paddock selection is a form of biological control.
- Using crops and varieties with resistance to the specific disease is also important. Other biological products are not necessarily available for disease control.

6. Responsible fungicide use:

- Synthetic pesticides are generally used only as required and often only at specific times in a disease lifecycle.
- Fungicides applied as protection ahead of conditions that are conducive to disease (e.g. sustained rainfall) may reduce total fungicide usage. Timing is critical with foliar fungicides, and may be more important than rate used. Protection is better than cure, because once the disease is established in the canopy, there is an internal source of infection that is difficult, or even impossible, to control with later fungicide applications. [6]

6 Australian Pulse Bulletin. Chickpea: integrated disease management. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/idm-strategies

9.4 Risk assessment

Prediction of likely damage from a chickpea disease can be used at the paddock, whole farm, regional, state or national level. The choices of variety and disease management options are some of the factors determining risk.

Knowledge of your paddock, its layout (topography), soil parameters, and cropping history will help you to assess the level of risk.

9.4.1 Steps in risk assessment

1. Identify factors that determine risk:

- **Pathogen**. Exotic *v.* endemic; biotypes, pathogenicity, survival and transmission, amenable to chemical management.
- **Host**. Host range; varietal reactions, vulnerability. Does susceptibility change with growth stage?
- **Environment**. Weather dependency, interactions with nutrition, herbicides, other diseases, agronomic factors, e.g. planting depth, row spacing, no-tillage, soil conditions.
- **Risk management**. Access to components of management plan; ease of implementing plan; how many options; cost of implementation.

2. Assess level of factors:

- **Pathogen**. Level of inoculum, dirty seed, aggressiveness of isolate, weed hosts prevalent in paddock or nearby, paddock history.
- **Host**. How susceptible, nutritional status, frost susceptibility, herbicide susceptibility.
- **Environment**. Length of season; likelihood of rain, drought, waterlogging, irrigation; availability of spray gear; paddock characteristics; herbicide history.
- **Risk management**: Not yet considered; plan being developed; plan in place?

3. What risk level is acceptable?

- **High**. Grower is prepared to accept substantial yield loss because potential returns are high and financial situation sound; crop failure will not affect rotation or other components of farming system.
- **Low**. Grower needs cash flow and cannot afford to spend much or lose the crop; failure seriously affects farming system.

9.4.2 Paddock selection

The selection of the most appropriate paddock for growing chickpeas involves consideration of several important factors, some of which are related to the modes of survival and transmission of pathogens such as *Ascochyta rabiei*.

1. Rotation:

- Develop a rotation of no more than one year of chickpea in four years.
- Plant chickpea into standing stubble of previous cereal to enhance crop height and reduce attractiveness of the crop to aphids (aphids may vector viruses).
- Consideration also needs to be given to previous crops that may host pathogens such as *Sclerotinia, Rhizoctonia*.
- *Ascochyta rabiei* is chickpea-specific, whereas *Botrytis cinerea* has a wide host range including sunflower, bean, pea, lentils and weeds (e.g. *Euphorbia* spp., groundsel and emufoot).

2. History of chickpea diseases:

- Previous occurrence of soil-borne diseases (Sclerotinia stem rot or Pratylenchus nematodes) constitutes a risk for subsequent chickpea crops for up to ten years.
- At least 500 m from the previous year's chickpea crop.

3. Weeds:

- Nearly all weeds host *Sclerotinia* spp.

- Some of the viruses affecting chickpea also have wide host ranges. Weeds, particularly perennial legumes, host viruses and their aphid and leafhopper vectors (e.g. Cucumber mosaic virus).

4. Herbicide history:

- Have triazine, sulfonylurea or other residual herbicides been applied in the last 12 months?
- The development of some diseases is favoured in herbicide-weakened plants. The presence of these herbicide residues in soil may cause crop damage and thus confusion over in-field disease diagnosis.

9.4.3 Regular crop monitoring

The two main diseases for which monitoring is necessary are Ascochyta blight and BGM. Following the monitoring process recommended for these diseases will provide the opportunity to assess the impact or presence of other diseases or plant disorders. To be effective, crop monitoring needs to include a range of locations in the paddock, preferably following a 'V' or 'W' pattern.

For Ascochyta blight

The initial symptoms will be wilting of individual or small groups of seedlings, or lesions on the leaves and stems of young plants, often in patches. Monitoring should commence 2–3 weeks after emergence, or 10–14 days after a rain event. This is because the initially infected seedlings soon die and symptoms are difficult to separate from other causes. Plant parts above the lesion may also break off, making symptoms difficult to detect. Timing is critical! After the initial inspection, subsequent inspections should occur every 10–14 days after a rain or heavy dew event. During dry periods, inspections should occur every two weeks. When monitoring, look for signs of wilting in upper foliage (the 'ghosting' phenomenon) or small areas of dead or dying plants, and if present, examine individual affected plants for symptoms of infection. This method will allow more of the crop to be inspected than a plant-by-plant check.

For Botrytis grey mould

Botrytis grey mould is more likely to occur in well-grown crops where there is canopy closure. The critical stage for the first inspection will be at the commencement of flowering and then regularly through the flowering period. Lesions occur on stems, leaves and pods, and flower abortion and drop can occur; a fluffy grey fungal 'bunch of grapes' growth develops on affected tissue. Normal pod-set will occur when daily temperature exceeds 15°C; BGM ceases to affect the plant once the maximum daily temperature exceeds ~28°C.

More regular crop monitoring may also be required if:

- high-risk situations exist such as non-optimal paddock selection
- shortened rotation
- immediately adjacent to last year's crop
- high disease pressure experienced last year
- a more susceptible variety is planted. [7]

For more information, see Chickpea disease management – Southern region.

9.4.4 Crop disease forecast

Crop disease forecast is a weekly, location- and season-specific estimate of risk for certain crop diseases during the cropping season and, for some diseases, offers management practices to avoid potential yield loss.

7 Australian Pulse Bulletin. Chickpea: integrated disease management. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/idm-strategies

Who this tool is for?

Grain growers in the southern and western grain-producing regions of Australia.

Questions this tool answers:

- What is the risk of crop loss due to this disease?
- What steps can I take to manage this disease?

Diseases and crops this tool covers:

Viral diseases:

- Barley yellow dwarf virus
- canola Beet western yellows virus
- lupin Bean yellow mosaic virus
- lupin Cucumber mosaic virus

Crops included in the risk forecasts:

- cereals (wheat, barley, oats and triticale)
- pulses (field pea, chickpea, faba bean, lentil)
- canola
- lupins

What this tool does

Crop disease forecast estimates the risk of certain crop diseases during the cropping season for specific locations. For some diseases, it offers management practices to avoid potential yield losses.

Each weekly forecast, where relevant, accounts for varietal resistance, chemical options, agronomic yield potentials and losses, agronomic constraints (frost and terminal drought), risks of spore showers, disease severity and disease-related yield losses.

Inputs

No inputs from growers are required.

Outputs

A disease forecast report may include, for each location:

- forecast risk in tables or maps
- estimated severity or spore maturity
- sowing guide
- rainfall to date and stubble moisture
- suggested management practices [8]

9.5 Ascochyta blight

Key points:

- Chickpea crops in southern Australia are being hit by a more virulent strain of the damaging ascochyta blight.
- Pulse pathologists in Victoria and South Australia have noted a marked decline in the resistance of several varieties of chickpeas, with varieties previously rated as moderately resistant performing like susceptible lines.
- It is imperative all chickpea seed should be treated with a thiram based fungicide to prevent seed transmission of AB on to the emerging seedlings in 2017.

8 Climate Kelpie. Crop disease forecast. GRDC, http://www.climatekelpie.com.au/manage-climate/decision-support-tools-for-managing-climate/crop-disease-forecast

- Susceptible crops will require regular vegetative and reproductive foliar fungicide sprays every 2 to 3 weeks.
- Moderately susceptible crops will require 3 to 4 strategic fungicide sprays during the season ahead of rain fronts, the sprays offering 2-3 weeks protection against infection. [9]

Ascochyta blight, caused by the fungus *Phoma rabiei* (formerly *Ascochyta rabiei*), is a serious disease of chickpeas in Australia. The fungus is different from the species of ascochyta that infects faba beans, lentils and field peas. The fungus can infect all above-ground parts of the plant and is most prevalent when cool, cloudy and humid weather occurs during the crop season.

Chickpeas were an important part of southern farming systems crop rotations until the late 1990s. They provided both economic and sustainability benefits to growers throughout southern Australia until widespread outbreak of Ascochyta blight in 1998. All cultivars grown at this time were susceptible to the disease and as a result of the epidemic, the area sown reduced from 150,000 hectares to less than 10,000 hectares within two years. [10]

Ascochyta blight is now considered to be endemic in most growing regions. Unlike some insect control strategies, there is no economic threshold for ascochyta. Management strategies are aimed at preventing the occurrence of disease and limiting its spread. [11]

Management and control of Ascochyta blight is the important factor in determining the viability of chickpea production. The first variety with improved Ascochyta blight resistance, Genesis™ 508, became available in 2005, Genesis™ 090 followed it in 2006, and others like Genesis™ 509 follow in 2008. The large kabuli varieties Almaz[⟡] and Nafice[⟡] became available in 2006. They along with all older varieties will require strategic fungicide management through the application of well-timed protective foliar fungicides. Disease levels will vary according to season, rotation history of the paddock and its surrounds, stubble management, seed hygiene, sowing time and timing of fungicide applications. [12]

Ascochyta blight is managed through crop rotation, hygiene, seed treatment, prophylactic fungicide application and growing varieties with improved resistance. All growers and advisers need to regularly inspect their crops from emergence, through flowering, right up to plant maturity. Inspections should be undertaken 10–14 days after rain events, when new infections will be evident as lesions on plant parts. [13]

Changes in Ascochyta blight—Southern region

During 2015 at Curyo (southern Mallee), despite the relatively dry season, in early August a significant outbreak of Ascochyta blight was observed in a kabuli chickpea trial (Figure 4). Symptom assessment indicated that this isolate of ascochyta was different from those observed previously in Victorian trials, having virulence on resistant lines such as Genesis™ 090 and PBA Slasher⟡. From the results in this trial, there appears to be some differences in resistance to this isolate with CICA1454 showing fewer symptoms and PBA Striker⟡ being significantly affected (Figure 4). Unfortunately, due to the dry finish to the season, and low yield potential it was impossible to assess the impact of the disease on grain yield. Despite being affected by the disease Genesis™ 090 was the highest yielding (0.5 t/ha) line at Curyo (yield data not shown).

The isolate from this trial was provided to pathologists and has been compared with other new isolates from SA. Results show that Genesis™ 090 has a susceptible

9 Jenny Davidson (2016). Personal communication.

10 I Pritchard (2000). Managing Ascochyta blight. Journal of the Department of Agriculture, WA, Series 4. Vol 41. Article http://researchlibrary.agric.wa.gov.au/cgi/viewcontent.cgi?article=1010&context=journal_agriculture4

11 K Moore, M Ryley, G Cumming, L Jenkins. (2015) Chickpea: Ascochyta blight management. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/ascochyta-blight

12 Pulse Australia (2007) Chickpeas in South Australia and Victoria. http://www.pulseaus.com.au/storage/app/media/crops/2007_Chickpeas-SA-Vic.pdf

13 JA Davidson, D Hartley, M Priest, M Krysinska-Kaczmarek, A McKay, ES Scott, (2009) A new species of Phoma causes Ascochyta blight symptoms on field peas (*Pisum sativum*) in South Australia. Mycologia, 101(1), 120-128.

reaction to the Victorian isolate but not to the other two isolates. Ambar⍉, Neelam⍉ and Genesis™ 079 demonstrated resistance to all three isolates; Almaz⍉ had a moderate reaction to the new SA isolate but a good level of resistance to the other two isolates; PBA Maiden⍉ had a moderate reaction to all three isolates; Kalkee, PBA Monarch⍉, and PBA Slasher⍉ have a moderate to susceptible reaction to all three isolates while PBA Striker⍉ had a susceptible reaction to all three isolates (Figure 4). These results still need to be confirmed with field data (Table 4). [14]

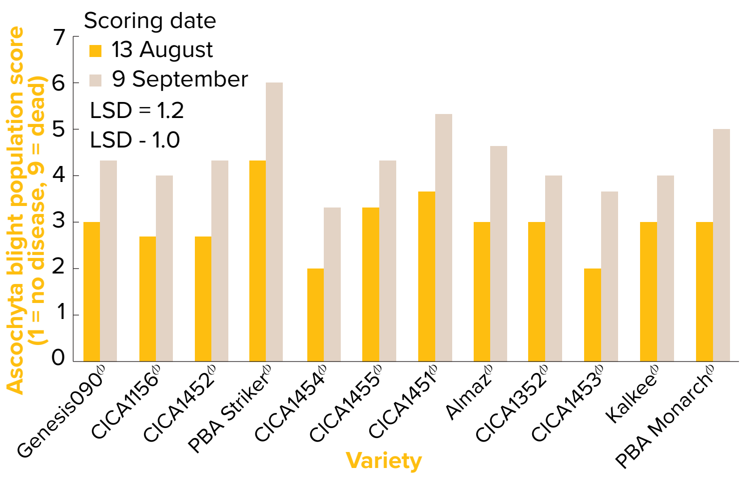

Figure 4: *Ascochyta blight score in the medium sized kabuli chickpea variety trial at Curyo 2015, recorded August 13 and September 9, 2015.*

Source: Brand *et al* via GRDC

Economic importance

The widespread occurrence of this disease in 1998 had a negative impact on the chickpea growing industry. To successfully grow varieties with an ascochyta disease rating less than moderately resistant, foliar fungicides need to be applied throughout the growing season to avoid serious yield losses. Varieties rated as moderately resistant (such as PBA Slasher⍉ and Genesis™ 509) still require at least one fungicide at early pod-set, but the risk of yield loss is minimal. When selecting varieties, the added cost of fungicide applications needs to be considered before selecting and growing susceptible to moderately resistant varieties. [15]

9.5.1 Varietal resistance or tolerance

With the spread of the new strain of ascochyta blight, the resistance ratings of all chickpea varieties have been updated (Table 4).

14 J Brand, M Rodda, G Rosewarne, A Delahunty, R Kimber, J Davidson, K Hobso (2016). Your future pulse—pulse breeding and agronomy update. GRDC Update Papers 19 July 2016, https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/07/Your-future-pulse-pulse-breeding-and-agronomy-update

15 AgVic (2008) Ascochyta blight of chickpea. Agriculture Victoria, Ag Note AG1186 August 2008 Updated August 2016, http://agriculture.vic.gov.au/agriculture/pests-diseases-and-weeds/plant-diseases/grains-pulses-and-cereals/ascochyta-blight-of-chickpea

Table 4: *Varietal resistance of chickpeas to the new strain of ascochyta blight.*

Chickpea variety	Ascochyta blight resistance rating rating (old rating in brackets)
Desi	
Ambar⍉	MS (R)
Genisis 509	MS (R)
Howzat⍉	S (S)
Neelam⍉	MS (R)
PBA HatTrick	S (MR/R)
PBA Maiden	S (MR)
PBA Slasher	MS (R)
PBA Striker	S (MR)
SONALI	S (MR)
Kabuli	
Almaz⍉	MS (MS)
Genisis 079	S (R)
Genisis 090	MS (R)
Genisis 114	S (S)
Kalkee	MS (MS)
PBA Monarch	S (MS)

Source: SA Sowing Guide 2017.

Before the outbreak of the new strain of ascochyta blight, there were resistant varieties available. Varieties differed in their resistance and/or tolerance to Ascochyta blight (Table 5 and Figure 5).

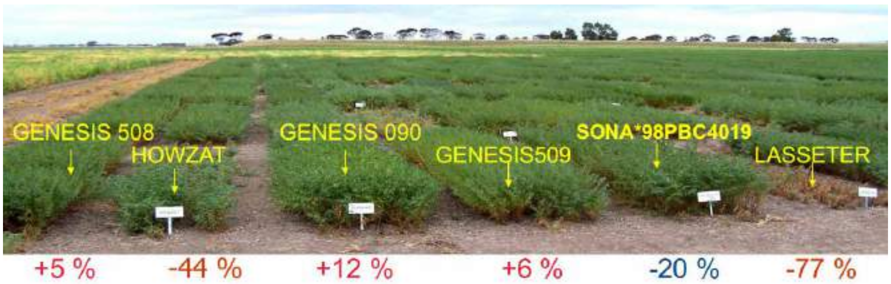

Figure 5: *Varietal response to Ascochyta blight in 2003 Horsham trials. Trials had no treatment with fungicide.*

Source: Hobson et al via University of Sydney

Table 5 estimates gross margins for chickpeas with ascochyta-susceptible versus ascochyta-resistant varieties. Fungicide costs are based on eight applications at $20/ha per application for the susceptible variety versus one for the resistant variety. Assuming variety yields are the same, desi gross margin of $130 versus $270/ha may be achieved from a 1.5 t/ha grain yield. A $180 versus $420/ha return could be obtained from a kabuli yield of 1.0 t/ha. If choosing a variety susceptible to Ascochyta blight, growers should consider kabuli production in preference to desi where conditions are suitable. [16]

16 Pulse Australia (2007) Chickpeas in South Australia and Victoria. http://www.pulseaus.com.au/storage/app/media/crops/2007_Chickpeas-SA-Vic.pdf

Table 5: *Estimated desi and kabuli returns. NOTE: these calculations are NOT based on varietal tolerance to the new Ascochyta blight strain.*

Grain yield (t/ha)	Grain price ($/t)	Fungicide cost Susceptible variety ($/ha)	Fungicide cost Resistant variety ($/ha)	Other costs All varieties ($/ha)	Gross margin Susceptible variety ($/ha)	Gross margin ($/ha) Resistant variety ($/ha)
Desi						
0.5	300	160	20	160	-170	-30
1.0	300	160	20	160	-20	120
1.5	300	160	20	160	130	270
2.0	300	160	20	160	280	420
Kabuli						
0.5	500	160	20	160	-70	70
1.0	500	160	20	160	180	420
1.5	500	160	20	160	430	570
2.0	500	160	20	160	680	820

Source: Pulse Australia

VIDEOS

1. Grains Research Updates 2015 – Ascochyta: Pathogen changes and management.

The national chickpea program continues its commitment to re-establishing the chickpea industry in south-east Australia. A number of international lines with excellent resistance to Ascochyta blight have been released and were the first resistant varieties available. All releases require less fungicide support than current options, but still require one spray at podding to produce good quality seed. Resistant varieties offer growers greater yield security. However, fungicides are still likely to be required to minimise yield losses through pod and seed infection. Seed infected by Ascochyta blight can be small and blemished. It may also attract lower returns through downgrading. Sowing diseased seed sets back crop prospects from the outset. [17]

9.5.2 Damage caused by disease

The high-risk and increased cost of controlling Ascochyta blight in a susceptible variety often make desi chickpea production unprofitable but higher value kabuli types remain profitable. [18] Unlike some insect control strategies, there is no economic threshold for Ascochyta. Management strategies are aimed at preventing the occurrence of disease and limiting its spread. [19]

9.5.3 Symptoms

Phoma rabiei infects the leaves, stems and pods of chickpea plants, causing tan/brown, rounded lesions on affected plant parts. This disease is usually first noticed in late winter when small patches of blighted plants appear throughout the paddock (Photo 1). Usually the first symptoms are the wilting of individual or small groups of seedlings. Plants appear as if premature haying off has occurred. Initially Ascochyta blight appears on the younger leaves as small water-soaked pale spots. These spots rapidly enlarge under cool and wet conditions, joining with other spots on the leaves and blighting the leaves and buds. [20]

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17 Pulse Australia (2007) Chickpeas in South Australia and Victoria. http://www.pulseaus.com.au/storage/app/media/crops/2007_Chickpeas-SA-Vic.pdf

18 L McMurray, J Brand, J Davidson, K Hobson, M Materne, (2006, September). Economic chickpea production for southern Australia through improved cultivars and strategic management to control Ascochyta blight. In Proceedings of 13th Australian Agronomy Conference (p. 65).

19 K Moore, M Ryley, G Cumming, L Jenkins. (2015) Chickpea: Ascochyta blight management. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/ascochyta-blight

20 AgVic (2008) Ascochyta blight of chickpea. Agriculture Victoria, Ag Note AG1186 August 2008 Updated August 2016, http://agriculture.vic.gov.au/agriculture/pests-diseases-and-weeds/plant-diseases/grains-pulses-and-cereals/ascochyta-blight-of-chickpea

Photo 1: *Wilting of individual or small groups of seedling.*

Source: Pulse Australia

Ascochyta leaf ghosting symptoms may appear 4–7 days after rainfall or heavy dew (Photo 2). [21]

Photo 2: *Ghosting symptoms of chickpea.*

Source: Pulse Australia

Lesions usually begin as a pale green-yellow discolouration on leaves and stems and progress into small round lesions with dark-brown margins and pale grey to tan sunken centres (Photo 3). Note the concentric circles of brown-black dots in the centre of the lesions. These small black spots (pycnidia), or fruiting bodies are unique to Ascochyta blight. Pycnidia, less than 1 mm in diameter, can be seen in the affected areas. Pycnidia are also present in stem lesions. In severe cases of infection the entire plant dries up suddenly.

21 K Moore, M Ryley, G Cumming, L Jenkins. (2015) Chickpea: Ascochyta blight management. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/ascochyta-blight

Photo 3: *Lesions on chickpea leaves caused by Ascochyta blight*

Source: Pulse Australia

Lesions on stems at first tend to be oval shaped, with brown centres and a darker margin. Elongated lesions can often form and girdle the stem (Photo 4). The stem may die and break off. Regrowth may occur from the broken stem. Affected areas on the pods tend to be round, sunken, with pale centres and dark margins.

Photo 4: *Stem lesion of chickpea leading to girdling and breakage of stem.*

Source: Pulse Australia

- Leaf lesions: Lesions usually begin as a pale green-yellow discolouration on leaves and stems and progress into small round lesions with dark-brown margins and pale grey to tan sunken centres. Towards the centre of the lesion, fruiting bodies called pycnidia develop (appearing as black specks), often in concentric rings. These pycnidia produce spores, which spread on wind-borne stubble and/or water (rain splash) to infect other plants. Note the concentric circles of brown-black dots in the centre of the lesions. These are the pycnidia or fruiting bodies that are unique to Ascochyta blight. Ascochyta leaf ghosting may appear 4–7 days after infection following rainfall or heavy dew.

- Stem lesions: Lesions on stems at first tend to be oval shaped, with brown centres and a darker margin. Lesions often girdle the stems of the plant, causing them to weaken and subsequently break off.
- Pod lesions: Pod lesions are similar in appearance to leaf lesions. They lead to infection of the seed. DO NOT keep planting seed from any crop that has been identified as having Ascochyta blight (Photo 5). [22]

Photo 5: *Pod lesions look similar to leaf lesions and lead to infection of the seed (left). Infected pod with a large lesion containing fungal fruiting bodies (right).*

Source: Pulse Australia and CropPro

The fungus can penetrate the pod and infect the seed. Pod lesions are similar in appearance to leaf lesions. Severe pod infection usually results in reduced seed set and infected seed. When infected seeds are sown, the emerging seedlings will develop dark-brown lesions at the base of the stem. Affected seedlings may collapse and die.

9.5.4 Conditions favouring development

Initial crop infection is due to the introduction of either infected planting seed or from movement of infected trash by wind, machinery or animals. Spores of the fungus can survive for a short time on skin, clothing and machinery. Subsequent in-crop infection and spread occurs when inoculum is moved higher in the canopy or to surrounding plants by wind or rain splash during wet weather. The disease spreads during cool, wet weather from infected plants to surrounding plants by rain splash of spores. This creates large blighted patches within crops. Pycnidia produce spores, which infect other plants through wind-borne stubble and/or water (rain splash). There are no other known hosts of *Phoma rabiei* in Australia. [23,24]

Ascochyta blight-infected stubble blown about during and after harvest is a major cause of short–medium-distance dispersal (metres to kilometres) along with movement of infected trash by water, machinery or animals. Spores of the fungus can survive a short time on skin, clothing and machinery.

22 K Moore, M Ryley, G Cumming, L Jenkins. (2015) Chickpea: Ascochyta blight management. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/ascochyta-blight

23 K Moore, M Ryley, G Cumming, L Jenkins. (2015) Chickpea: Ascochyta blight management. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/ascochyta-blight

24 K Moore, M Ryley, G Cumming, L Jenkins. (2015) Chickpea: Ascochyta blight management. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/ascochyta-blight

Ascochyta blight can increase rapidly on volunteer chickpeas if wet weather occurs during spring–summer–autumn. Paddocks with chickpea stubble should be regarded as a source of inoculum even if Ascochyta blight was not observed in last season's chickpea crop. The pathogen can survive at least three years in the paddock.

Ascochyta blight can develop over a wide range of temperatures (5–30°C) and needs only 3 hours of leaf wetness to infect (Figure 6). However, the disease develops fastest when temperatures are 15–25°C and relative humidity is high (the longer relative humidity remains high, the more severe will be the infection).

Subsequent in-crop infection occurs when spores are moved higher in the canopy or to surrounding plants by rain splash during wet weather. Multiple cycles of infection will occur during the growing season whenever environmental conditions are favourable.

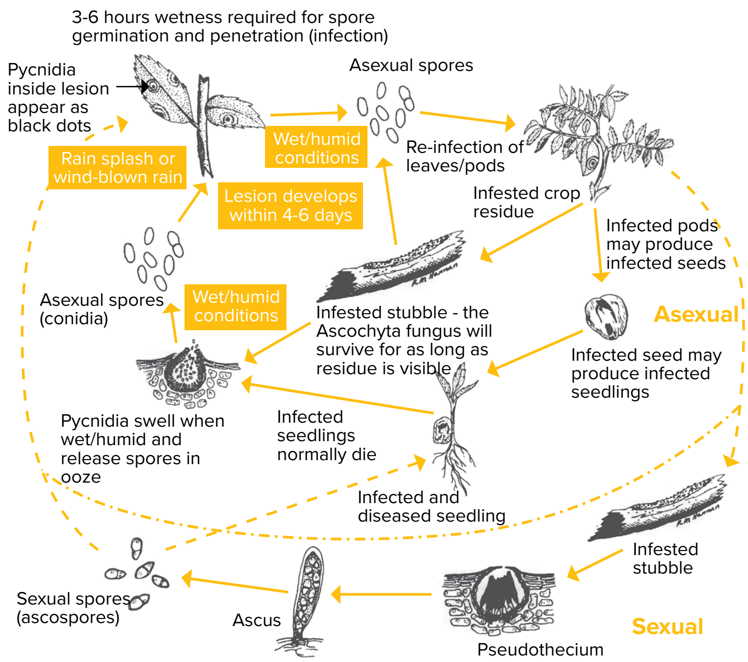

Figure 6: *Life cycle of Ascochyta blight pathogen. Note: Only the asexual phase is known to occur in Australia at this time.*

Drawings by RM Hannan (Can. J. Pl. Path. 19:215-224, 1997)

FAQ

9.5.5 Management of disease

Monitoring:

- When inspecting crops, look for signs of wilting in upper foliage and small areas of dead or dying plants.
- Check in a range of locations across the field following a 'V' or 'W' pattern.
- Spend at least 1 to 2 hours inspecting each crop for Ascochyta blight.
- Ensure good hygiene when moving between crops and farms.

Take extra care when inspecting crops that are growing:

- under centre pivot or lateral-move irrigators
- from seed whose ascochyta status is unknown
- from seed that was not treated with a registered fungicide seed dressing. [25]

25 K Moore, M Ryley, G Cumming, L Jenkins. (2015) Chickpea: Ascochyta blight management. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/ascochyta-blight

If ascochyta is suspected

If ascochyta is suspected mark the spot and take samples for diagnosis. DO NOT enter other chickpea paddocks wearing the same clothing. All other chickpea crops on the property need to be inspected for Ascochyta blight. Be sure to follow the hygiene practices outlined below:

- Place samples of suspected ascochyta-infected plants into a plastic bag then seal the bag and keep the samples cool.
- Suspect samples should be referred to a plant pathologist or agronomist familiar with the disease for identification.
- Unnecessary movement within a suspected ascochyta-infected crop should be avoided until the sample has been fully assessed.
- Most importantly, do not visit other chickpea crops until all clothing has been disinfected or changed and machinery has been washed of all plant material and dirt. [26]

Hygiene

The spores of ascochyta can adhere to clothing, machinery, vehicles, people and animals when moving through infected paddocks, so hygiene is a vital component of IDM when ascochyta is found in a crop. Wear waterproof pants, overboots or rubber gum boots when entering a suspected infected paddock, then decontaminate immediately after exiting.

- Farmers and advisers should take precautions to prevent spreading Ascochyta blight via clothing, footwear and vehicles.
- The recommended protocol is for clothing to be washed, changed or disinfected when moving between chickpea paddocks.
- Wash boots in a mixture of 10% bleach and 90% water solution or methylated spirits upon leaving an infected chickpea crop.
- Clothing must be machine-washed in hot water before being worn when entering another chickpea crop.
- Extra care should be taken to remove soil and plant material from boots and vehicles.
- Hands and arms should be washed in warm soapy water or a suitable disinfectant.
- The use of heavy-duty plastic bags to cover boots and legs is a common practice when checking crops. After inspecting the crop, remove these plastic covers and place them in another bag and seal. Use another set of covers if you need to enter another chickpea crop.
- Farmcleanse® can be used to clean equipment. [27]

During harvest

Harvest ascochyta-free paddocks before infected paddocks and preferably use your own harvester. Do not run the straw spreaders when harvesting, which will reduce the spread of small pieces of ascochyta-infected stem and pods.

Thoroughly clean and decontaminate all machinery associated with harvesting in a well-defined and identifiable area before moving to another paddock or property.

Post-harvest

All grain harvested from an ascochyta-infected paddock should be transported off farm to receival sites in well-sealed trucks. If kept for a period on-farm it should be stored in well-sealed and labelled silos which must be thoroughly cleaned after the grain has been removed. Grain harvested from an ascochyta-infected crop must not be retained as planting seed for other crops. Consideration may be given to

26 K Moore, M Ryley, G Cumming, L Jenkins. (2015) Chickpea: Ascochyta blight management. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/ascochyta-blight

27 K Moore, M Ryley, G Cumming, L Jenkins. (2015) Chickpea: Ascochyta blight management. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/ascochyta-blight

incorporation of infected crop residues by the use of off-set discs immediately after harvest to enhance the rate of breakdown. Chickpea volunteers in the infected paddock, along fence lines and near sheds must be controlled. Chickpeas should not be grown in or adjacent to an ascochyta-infected paddock for at least three years. [28]

Control

Follow the principles of IDM, which include:

- crop rotation and paddock selection
- clean seed and fungicide seed dressings
- regular crop monitoring
- strict hygiene on and off farm
- strategic use of foliar fungicides.

Note: Chickpea seed dressings only protect the emerging seedling from seed-borne ascochyta and seed-borne botrytis. Seed dressings will not protect the emerged seedling from rain-drop splashed ascochyta or wind-borne botrytis. [29]
See Section 3 Planting, 3.2 Seed treatments for more information.

Sowing date:

- With Ascochyta blight resistant varieties, sow at traditional sowing dates. Delayed sowing is not necessary.
- With Ascochyta blight susceptible varieties, delayed sowing has been a most important strategy for Ascochyta blight management. It reduces the duration of exposure of chickpea seedlings to Ascochyta blight spores. It will not help though if self-sown chickpeas are nearby. Be aware that delayed sowings can result in lower yields due to increased risks of dry finishes and high temperatures during podding.
- In all varieties, sowing too early can produce poor early pod-set if flowering is in a colder period. [30]

Harvest timing

Harvest as early as possible to minimise Ascochyta blight infection on seed and potential seed downgrading. The damage from the disease is usually more severe when crops are harvested late. Harvest losses, seed splitting and downgrading quality can be substantial if chickpea is harvest is below 12%. [31]

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28 K Moore, M Ryley, G Cumming, L Jenkins. (2015) Chickpea: Ascochyta blight management. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/ascochyta-blight

29 K Moore, M Ryley, G Cumming, L Jenkins. (2015) Chickpea: Ascochyta blight management. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/ascochyta-blight

30 W Hawthorne, J Davidson, L McMurray, K Lindbeck, J Brand (2012) Chickpea disease management strategy—Southern region. Pulse Australia, Southern Pulse Bulletin PA 2012 #08, http://pulseaus.com.au/storage/app/media/crops/2012_SPB-Chickpea-disease-management.pdf

31 W Hawthorne, J Davidson, L McMurray, K Lindbeck, J Brand (2012) Chickpea disease management strategy—Southern region. Pulse Australia, Southern Pulse Bulletin PA 2012 #08, http://pulseaus.com.au/storage/app/media/crops/2012_SPB-Chickpea-disease-management.pdf

IN FOCUS

Management options for minimising the damage by Ascochyta blight (*Ascochyta rabiei*) in chickpea (*Cicer arietinum L.*)

Ascochyta blight, a fungal disease caused by *Ascochyta rabiei* is the major constraint for chickpea production worldwide. Current cultivars only possess partial resistance to the pathogen, and this level of resistance can breakdown easily because the pathogen is highly variable due to potential for sexual recombination. The development of IDM is the key for successful chickpea production. The use of Ascochyta blight-free seed and seed dressing with effective fungicides reduces the probability of transmitting seed-borne disease to the seedlings. Deep-burying or burning of chickpea stubble minimises stubble-borne inoculum. One to two years of non-host crops for warm and wet areas and 3–4-year crop rotation for cold and dry areas are required to reduce the levels of stubble-borne inoculum. The use of field isolation and sowing chickpea at a distance from previous chickpea crops will reduce the density of airborne ascospores released from infected debris. Optimum sowing date, deep sowing, optimizing plant density, balanced nutrition, and alternative sowing patterns should be considered as a means of reducing Ascochyta blight pressure wherever possible. Sprays at seedling stage or before the occurrence of infection are crucial in short-season areas or where ascospores are the major sources of inoculum. Chickpea growers are strongly encouraged to adopt an integrated approach that combines all agronomic options, including cultivar selection, if they are to manage this disease economically and effectively. [32]

MORE INFORMATION

Chickpea: Ascochyta blight management.

9.5.6 Ascochyta blight management in kabuli

Paddock selection

Keep at least a three-year break between chickpea crops in the same paddock. Equally importantly, sow new chickpea crops at least 500 m from any paddock (yours or your neighbours) in which chickpea was grown in the previous season. Ascochyta spores from infected chickpea stubble from the previous season are released in mid-winter and can be blown hundreds of metres, or even kilometres. Small pieces of infected chickpea trash (leaf, pod and stem) may be blown considerable distances during harvest and may also be moved about by winds throughout the summer and autumn. It is important to consider the risks from wind-blown trash prior to the break of the season and wind-borne spores after crop emergence when selecting paddocks to sow to chickpea. [33]

Seed

Test your seed for germination and Ascochyta blight infection. Do not sow seed if the ascochyta infection level is above 0.25%. All kabuli seed should be treated with a fungicide seed dressing; this will reduce the transmission of seed-borne fungal infections and also help to protect the emerging seedling from soil-borne pathogens and seedling rots. Seed testing and seed dressing are complementary: seed testing ensures that seed with an unacceptably high level of infection is not being sown

32 YT Gan, KHM Siddique, WJ MacLeod, P Jayakumar (2006) Management options for minimizing the damage by Ascochyta blight (Ascochyta rabiei) in chickpea (*Cicer arietinum L.*). Field Crops Research, 97(2), 121-134.

33 K Regan (2016) Production packages for kabuli chickpea in Western Australia—post planting guide. DAFWA, https://www.agric.wa.gov.au/chickpeas/production-packages-kabuli-chickpea-western-australia-post-planting-guide

while seed dressing reduces, but does not eliminate, seed-borne infection. Seed dressing highly infected seed reduces the level of transmission, but may still result in high levels of initial infection of the emerged crop. [34]

Fungicide timing

Where crops of Almaz⁽ᶦ⁾ and KALKEE⁽ᶦ⁾ have been established following the above recommendations, growers should budget for two or three strategic fungicide sprays (chlorothalonil 720 g/L applied at 1.0–2.0 L/ha). This is a significant improvement over the regular spray schedule (every three to four weeks) previously recommended.

The fungicide spray is required four weeks after emergence (chlorothalonil 720 g/L applied at 1.5 L/ha). This early prophylactic spray is required to contain the spread from any Ascochyta blight infections resulting from wind-blown spores from last year's stubble, seed-borne infections or infected trash that has been carried into the paddock. The level of infection that requires application of a fungicide spray this early in the crop's life is very low and is below the level that can be reliably identified, even by a person who has considerable experience in identifying this disease in field crops. Additionally, application of an early spray will protect the crop against wind-borne spores released from chickpea stubble during the two to three weeks following the spray application.

A second spray (chlorothalonil 720 g/L applied at 1.0–2.0 L/ha) is recommended at full flowering to protect the developing pods and minimise the risk of reduced quality. The rate of fungicide application depends on the level of Ascochyta blight infection detected in the crop prior to spraying. The high rate (2.0 L/ha) would be appropriate where Ascochyta blight can be easily identified in the crop and the low rate (1.0 L/ha) where only minor disease infection is evident after close inspection. If Ascochyta blight is not identified, even after close inspection of more than ten locations throughout the crop, a fungicide application may not be required at this time. A fungicide spray (chlorothalonil 720 g/L applied at 1.5–2.0 L/ha) may be required during pod-filling if Ascochyta blight becomes evident in the canopy during late flowering or podding. [35]

9.5.7 Foliar fungicide programs

Ascochyta blight has constrained chickpea production in Australia. Therefore, control strategies are required to prevent major crop losses. Field experiments in 1998 and 1999 showed that all the chickpea varieties grown commercially in Australia at that time were very susceptible to the disease. Fortnightly sprays with the fungicide chlorothalonil could effectively control epidemics but the additional cost significantly reduced profitability. The kabuli variety Kaniva was still profitable to grow but desi varieties were less profitable than alternative crops.

Further experiments were conducted throughout Australia in 1999, 2000 and 2001 to compare a range of fungicides and to determine the optimum rates and frequency of fungicide sprays. Chlorothalonil was superior to mancozeb and carbendazim. Fortnightly sprays of chlorothalonil controlled Ascochyta blight in all varieties; sprays every three weeks did not eliminate yield losses due to Ascochyta blight in susceptible varieties under high disease pressure. Low fungicide rates were less effective than maximum recommended rates when conditions favoured a severe epidemic.

Field experiments were established in 2002 and 2005 to compare new varieties with the older, susceptible varieties. The new varieties had significantly less disease than the older varieties and did not require fortnightly sprays. The best new varieties required fungicide sprays only at the podding stage in order to prevent pod and seed infection.

34 K Regan (2016) Production packages for kabuli chickpea in Western Australia—post planting guide. DAFWA, https://www.agric.wa.gov.au/chickpeas/production-packages-kabuli-chickpea-western-australia-post-planting-guide

35 K Regan (2016) Production packages for kabuli chickpea in Western Australia—post planting guide. DAFWA, https://www.agric.wa.gov.au/chickpeas/production-packages-kabuli-chickpea-western-australia-post-planting-guide

As more varieties with greater resistance become available, growers will need to apply fewer fungicides and the consequences of missing a fungicide spray will be less serious. However, variety specific management strategies still need to be developed to enable growers to tailor their control strategy to each variety susceptibility in order to minimise fungicide usage and maximise profits. [36]

IN FOCUS

Economic chickpea production for southern Australia through improved cultivars and strategic management to control Ascochyta blight.

Cultivars with improved resistance to Ascochyta blight became available in 2005, but the successful economic management of the disease needed to be demonstrated to growers. Experiments were sown at four locations over two seasons in southern Australia to assess fungicide (chlorothalonil or mancozeb) application timing and efficacy in controlling Ascochtya blight in cultivars varying in Ascochyta blight resistance. Resistant (R) cultivars were successfully grown with two or less fungicide applications during podding. Moderately resistant (MR), moderately susceptible (MS) and susceptible (S) cultivars always required at least three and up to nine fungicide applications to prevent yield loss. In all experiments the podding treatment of chlorothalonil had equivalent or greater grain yields than the mancozeb podding treatment. The use of resistant cultivars with one or two strategic foliar fungicide applications ensures chickpeas are a low risk, profitable option in medium rainfall (350–450 mm) cropping areas of southern Australia.

Experimental design

Field experiments were sown in 2004 and 2005 across south-eastern Australia to compare the effect of different fungicide regimes on Ascochtya blight foliar symptoms and grain yield of cultivars varying in levels of Ascochyta blight resistance. Trials were located at Turretfield (light clay over medium clay; annual rainfall (AR): 456 mm) and Hart (clay loam over heavy clay; AR: 399 mm), in the Mid North district of SA in 2004 and 2005. In 2005, trials were also located at Kalkee in the Wimmera (black cracking clay, AR: 450 mm) and at Beulah in the southern Mallee (calcareous sandy loam over medium clay, AR: 375 mm) of Victoria (Vic). Five fungicide regimes were compared in each experiment: Nil - no foliar fungicide sprays, Fortnightly application of chlorothalonil (Fn, 1440 gai/ha at SA sites and 750 gai/ha at Vic sites) applied every fourteen days from eight weeks after sowing till end of podding (six to nine sprays), Strategic application of chlorothalonil (St, three sprays at SA sites and four at Vic sites from eight weeks after sowing through to podding, single application at early podding of chlorothalonil (P°C), and mancozeb (PoM, 1650 gai/ha at SA sites and 750 gai/ha at Vic sites). The podding treatments at Turretfield in 2005 included a second application of the same chemical three weeks after the first. Cultivars varied across experiments depending upon seed availability and suitability for each district (Table 6). Nevertheless, all experiments included an Ascochyta blight S cultivar (Howzat⍉ - desi type) and R cultivars (Genesis™ 090 – kabuli type and Genesis™ 509 – desi type). Ascochyta blight ratings of other cultivars evaluated were Genesis™ 508 (desi type R), SONAL⍉ (desi type, MS) and Almaz⍉ (kabuli type MR). All treatments were replicated four times utilising either split plot (SA sites in 2004 and Vic. sites) or randomised complete block designs (SA sites in 2005).

36 TW Bretag, WJ MacLeod, RBE Kimber, KJ Moore, EJC Knights, JA Davidson (2008) Management of Ascochyta blight in chickpeas in Australia. Australasian Plant Pathology Society, Australasian Plant Pathology, 37(5), 486-497.

Desi chickpea cultivars were sown to achieve a plant density of 50 plants/m² and kabuli cultivars 35 plants/m². Seed was inoculated with Group N rhizobium and sown with suitable rates of fertiliser at appropriate sowing dates for chickpeas in each cropping region. All experiments were sown in paddocks not containing or not close to chickpeas or chickpea stubbles, and were artificially inoculated with infected stubble approximately between five and six weeks after emergence to induce significant Ascochyta blight disease pressure.

The effect of foliar fungicide application timing

A significant interaction between cultivar and fungicide timing for foliar Ascochyta blight infection occurred at all sites. In all experiments, the R cultivars Genesis™ 090, Genesis™ 508 and Genesis™ 509 showed lower levels of foliar symptoms in all treatments (score <2.5 or <22%) (Table 6). In contrast, Ascochyta blight severity in nil treatments of the S cultivar Howzat⁽ᵇ⁾ ranged from a score of 6.8–7.0 (Hart and Turretfield) in 2004 to greater than 9.0 and more than 90% infection in 2005 at Beulah, Turretfield, Hart and Kalkee. Disease ratings in nil treatments of MS (SONALI⁽ᵇ⁾) and MR (Almaz⁽ᵇ⁾) cultivars were intermediate with scores of 5.3 or 30–45% infection, respectively. However, these cultivars were only evaluated at sites with higher levels of disease intensity. Unlike the resistant cultivars, foliar symptoms in Howzat⁽ᵇ⁾, SONALI⁽ᵇ⁾ and Almaz⁽ᵇ⁾ were reduced substantially with Fn treatments. At sites with lower disease severity, the foliar disease level of Howzat⁽ᵇ⁾ was reduced with St treatments compared to the nil treatment but not to the level of the Fn treatment. The St treatment had no effect at sites where disease levels were high. This partial reduction in foliar disease also occurred in SONALI⁽ᵇ⁾ and Almaz⁽ᵇ⁾ with St, PoM and P°C treatments at most sites. Gan *et al*. (2006) suggested application of foliar fungicides at seedling stages for MR cultivars to minimise the impact of intense showers of ascospores. Our data indicates that while S and partial resistant (MS and MR) cultivars may require this early spray to minimise disease intensity, R cultivars did not.

The interaction between cultivar and fungicide timing for grain yield was significant in all experiments, except Hart in 2004 where dry and hot seasonal conditions during podding resulted in low grain yields. Grain yield was reduced at all other sites in nil treatments of Howzat⁽ᵇ⁾ compared to Fn treatments, with yield reductions ranging from 66 to 99% (Table 6). The grain yield of R cultivars was not always reduced in nil treatments when compared with Fn treatments. Where grain yield reductions in these cultivars did occur it was substantially lower than in all other cultivars, ranging from 0 to 37% in Genesis™ 090, 0 to 43% in Genesis™ 508 and 0 to 37% in Genesis™ 509. Nil treatments of SONALI⁽ᵇ⁾ and Almaz⁽ᵇ⁾ incurred yield loss in all experiments, ranging from 84 to 96% and 51 to 94%, respectively. The P°C and St treatments in Howzat⁽ᵇ⁾, SONALI⁽ᵇ⁾ and Almaz⁽ᵇ⁾ had substantially lower grain yields than those of the fortnightly treatments in all experiments. At sites with lower disease intensities such as Turretfield in 2004 and Beulah the St treatment reduced yield loss in Howzat⁽ᵇ⁾ by only 12 and 17% respectively, compared to the Fn treatment. However, higher disease intensity grain yield losses increased to 50 and 96% at Hart in 2005 and Turretfield in 2005, respectively. A similar pattern also occurred in SONALI⁽ᵇ⁾ for these treatments; however yield losses were generally less than for Howzat⁽ᵇ⁾. Grain yield loss of Almaz⁽ᵇ⁾ when compared with the Fn treatment ranged from 20 to 64% in the P°C treatment and 13 to 76% in the St treatment, significantly less than that incurred for Howzat⁽ᵇ⁾ and SONALI⁽ᵇ⁾. Grain yields were similar in the P°C and St treatment for Almaz⁽ᵇ⁾, whereas SONALI⁽ᵇ⁾ and Howzat⁽ᵇ⁾ generally had greater losses in the P°C treatment. This clearly indicates that Almaz⁽ᵇ⁾ has a higher level of Ascochyta blight resistance than Howzat⁽ᵇ⁾ and SONALI⁽ᵇ⁾ and would require fewer foliar fungicides sprays for successful production.

Table 6: *Effect of various foliar fungicide regimes on foliar Ascochyta blight severity (0–9 and 0–100% assessed during flowering/podding) and grain yield (t/ha) of chickpea cultivars varying in resistance to Ascochyta blight across South Australia and Victoria 2004 and 2005. # in () after Fn indicates total number of sprays applied for this treatment at that site*

		Foliar Ascochyta blight severity (1–9) or %						Grain yield (t/ha)					
Site	**Treat-ment**	**How-zat**	**Son-ali**	**Al-maz**	**Gen. 090**	**Gen. 508**	**Gen. 509**	**How-zat**	**Son-ali**	**Al-maz**	**Gen. 090**	**Gen. 508**	**Gen. 509**
Hart 2004 *(1-9)*	Nil	7.0			1.7	1.7	1,7	0.19			0.32	0.25	0.38
	PoM	6.9			1.4	1.9	1.9						
	P°C	6.9			1.4	1.4	1.4	0.14			0.32	0.25	0.35
	St	5.0			1.0	1.3	1.0	0.25			0.33	0.27	0.35
	Fn (6)	2.7			1.3	1.0	1.0	0.27			0.25	0.28	0.38
		lsd (P <0.01) = 0.99						NS					
T/ field 2004 *(1-9)*	Nil	6.8			2.2	1.8	1.6	0.70			1.77	1.58	1.89
	PoM	6.5			2.2	2.0	1.8	0.99			1.87	1.58	1.85
	P°C	6.5			2.4	1.3	1.8	1.07			1.90	1.60	1.73
	St	2.5			0.9	1.3	1.1	1.79			1.85	1.63	1.94
	Fn (6)	1.8			0.7	1.0	0.8	2.04			1.91	1.64	1.88
		lsd (P <0.01) = 0.94						lsd (P <0.05) = 0.23					
Beulah 2005 *(1-9)*	Nil	8.0	6.8		2.3		1.3	0.54	0.29		2.38		2.10
	PoM	7.5	7.0		2.3		1.8	0.46	0.16		2.50		2.02
	P°C	6.5	6.5		2.0		1.8	0.51	0.53		2.31		2.00
	St	3.8	3.3		1.0		1.0	1.96	1.94		2.76		2.32
	Fn (7)	1.8	1.8		1.0		1.0	2.35	2.75		2.77		2.30
		lsd (P <0.05) = 1.0						lsd (P <0.05) = 0.30					
Hart 2005 *(%)*	Nil	92	84	30	15	9	12	0.05	0.29	0.89	1.37	1.30	1.51
	PoM	76	61	29	21	10	14	0.20	0.68	1.09	1.52	1.39	1.61
	P°C	71	56	28	14	10	8	0.49	0.99	1.44	1.83	1.43	1.70
	St	61	44	20	11	12	8	0.89	1.23	1.54	1.77	1.58	1.74
	Fn (8)	25	21	11	9	8	5	1.89	1.84	1.8	1.81	1.62	1.85
		lsd (P <0.05) = 9						lsd (P <0.05) = 0.2					
Kalkee 2005 *(1-9)*	Nil	8.3	7.5	5.3	1.8	1.3	1.3	0.01	0.11	0.53	1.85	1.67	2.01
	PoM	8.3	7.8	5.0	1.5	1.5	1.3	0.03	0.18	0.58	1.80	1.60	1.87
	P°C	8.5	7.8	5.8	1.8	1.5	1.5	0.04	0.20	0.71	1.90	1.65	1.87
	St	8.3	7.0	4.3	1.3	1.0	1.5	0.18	0.44	1.37	2.16	1.72	1.95
	Fn (8)	2.3	1.8	1.3	1.0	1.0	1.0	1.82	1.68	1.71	2.19	1.74	2.05
		lsd (P <0.05) = 1.0						lsd (P <0.05) = 0.21					
T/field 2005 *(%)*	Nil	98	95	41	12	11	12	0.03	0.12	0.12	1.76	1.88	2.06
	PoM	99	90	41	14	11	12	0.07	0.18	0.22	1.87	1.96	2.34
	P°C	100	84	39	16	14	11	0.10	0.47	0.76	2.26	2.20	2.58
	St	90	64	46	12	14	9	0.10	0.98	0.47	2.41	2.62	3.04
	Fn (9)	29	28	12	10	8	9	2.34	3.16	1.91	2.79	3.32	3.28
		lsd (P <0.05) = 8						lsd (P <0.05) = 0.28					

Treatments P°C and St in Genesis™ 509 and Genesis™ 508 and the St treatment in Genesis™ 090 yielded the same as the Fn treatment in all experiments except for Turretfield in 2005. There was also no difference in grain yields in the P°C and Fn treatments of Genesis™ 090 at Turretfield in 2004 and Hart in 2005. However, grain yields were 17%, 13% and 19%

lower in the P°C treatment at Beulah, Kalkee and Turretfield in 2005, respectively. In Turretfield 2005 a second spray of chlorothalonil was used in the P°C spray regime due to an extended period of wet weather and an abnormally long flowering and podding period. Ascochyta blight pressure was particularly severe in this experiment and even MR cultivars were likely to have incurred a yield penalty even with the Fn treatment (Table 6). Botrytis grey mould (*Botrytis cinerea)* disease was also present at Turretfield in 2005 and pod infection and abortion across all cultivars was observed, potentially confounding results, despite two applications of procymidone (250 gai/ha). Results indicated that R cultivars, unlike all other cultivars evaluated, could be successfully grown with fungicide applications only during podding, apart from in situations of extreme disease severity such as those at Turretfield in 2005.

Efficacy of mancozeb and chlorothalonil fungicides as a podding spray

Chlorothalonil had greater efficacy than mancozeb when applied as a podding spray at some sites, particularly on cultivars rated MR or less to Ascochyta blight. At Hart in 2005, pod infection was greater in all cultivars (Table 7) in the PoM compared to the P°C treatment. Grain yields and grain weight were less in all cultivars except Genesis™ 509 (R) and Genesis™ 508 (R). Grain yields at Turretfield in 2005 for SONALI^Φ^ (MS), Almaz^Φ^ (MR) and Genesis™ 090 (R) and Beulah in 2005 for SONALI^Φ^ were significantly less in the PoM compared to P°C treatment. For all varieties at all sites grain yield in the PoM treatment were never significantly greater than P°C.

Table 7: *Comparison of chlorothalonil (P°C) and mancozeb (PoM) foliar fungicide treatments at podding on pod Ascochyta blight severity (0-100% assessed at maturity) and grain weight (g/100seeds) of chickpea cultivars varying in resistance to Ascochyta blight at Hart in South Australia, 2005.*

		Podding Ascochyta blight severity (%)						Grain weight (g/100 seeds)					
Site	**Treat-ment**	**How-zat**	**Son-ali**	**Al-maz**	**Gen. 090**	**Gen. 508**	**Gen. 509**	**How-zat**	**Son-ali**	**Al-maz**	**Gen. 090**	**Gen. 508**	**Gen. 509**
Hart 2005	P°C	75.0	58.8	11.3	9.3	4.0	13.5	20.1	17.5	44.5	33.9	15.7	15.7
	PoM	96.3	97.5	80.0	75.0	40.0	38.8	15.5	16.2	38.0	29.6	14.5	15.3
		lsd (P <0.05) = 23.3						lsd (P <0.05) = 1.6					

Conclusion

Cultivars with R to Ascochyta blight can be successfully grown under high disease pressure in southern Australia with only one or two fungicide applications during podding. This will allow growers to economically produce chickpeas again. Under the same conditions MS cultivars will require between three and nine fungicides to avoid severe yield loss. Cultivars with MR will require fungicide sprays prior to flowering in addition to podding sprays to prevent significant yield loss. This is likely to limit production of MR cultivars in southern Australia to the higher valued large seeded kabuli types, which are best adapted to the medium to high-rainfall growing areas. [37]

Differing spray programs have been developed based on each variety's ascochyta rating. Chickpea ascochyta fungicides are protectants only—unlike wheat stripe rust fungicides—they have no systemic or kick-back action, and they will not eradicate an existing infection. To be effective they must be applied before infection i.e. before rain. The key to a successful ascochyta spray program is regular monitoring combined with timely application of registered fungicides (Table 8 and 9). [38]

37 L McMurray, J Brand, J Davidson, K Hobson, M Materne, (2006, September). Economic chickpea production for southern Australia through improved cultivars and strategic management to control Ascochyta blight. In Proceedings of 13th Australian Agronomy Conference (p. 65)..

38 K Moore, M Ryley, G Cumming, L Jenkins. (2015) Chickpea: Ascochyta blight management. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/ascochyta-blight

Table 8: *Foliar fungicides for the control of Ascochyta and Botrytis grey mould.*

Active ingredient	Example trade name	Rate	
		Ascochyta blight	Botrytis grey mould
Chlorothalonil (720 g/L)	Crop Care Barrack® 720# Barrack Betterstick® # Nufarm Unite® 720#	1.0–2.0 L/ha	Not registered
Mancozeb (750 g/kg)	Dithane™ Rainshield™	1.0–2.2 kg/ha	1.0–2.2 kg/ha
Mancozeb (420 g/L)	Penncozeb® SC	1.8– 3.95 L/ha	Not registered
Carbendazim (500 g/L)	Spin Flo®	Not registered	500 mL/ha

These are the only registered chlorothalonil products. It is an offence to use any other product. Refer to current product label for complete 'Direction for use' prior to application.

Source: Pulse Australia

Table 9: *Chickpea foliar fungicides for the southern region.*

	Company	Active Ingredient	Rate	Av.Cost($/ha) [1]	Chickpeas	
					Botrytis	Ascochyta blight
1. Barrack® 2. Unite 720® 3. Bravo® 4. Bravo Weather Stik®	1.Crop Care 2.Nufarm 3,4.Syngenta	Chlorothalonil 720 g/L	1.0-2.0 L/ha	$16.70-$33.42	-	✓P, RA
1. Bavistin® 2. Carbend® 3. Spinflo® 4. Howzat⁽ᵈ⁾®	1. Crop Care 2,3.Nufarm 4. Farmoz	Carbendazim 500 g/L	500 mL/ha	$10.00-$13.50	✓	-
1. Penncozeb 420SC®	1.Nufarm	Mancozeb 420 g/L	1.8-3.5 L/ha*	$11.10-$26.60	-	✓
1. Penncozeb 750 DF® 2. Dithane Rainshield® 3. Mancozeb®	1.Nufarm 2.Dow 3.Griffin	Mancozeb 750 g/kg	1.0-2.2 kg/ha*	$7.60-$16.70	✓	✓
1. Polyram®	1.Nufarm 700 g/kg	Metiram	2.0 kg/ha		✓P E, RA	-
1. Sumisclex®	1.Sumitomo	Procymidone 500 g/L	500 ml/ha	$16.40-$33.80	RA	-

Note: Observations in 2010 Tamworth trials indicated that the natural resistance all plants have to pathogens and pests is compromised when plants are stressed from waterlogging and that this reduced the ability to manage ascochyta with a fungicide strategy that worked in less stressed plots. In a season when repeated cycles of infection occur, even MR varieties can have yield-reducing levels of disease. [39]

Source: NDSU

39 K Moore, M Ryley, G Cumming, L Jenkins. (2015) Chickpea: Ascochyta blight management. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/ascochyta-blight

Fungicide information

Chlorothalonil

Pulse Australia's application has been denied by the Australian Pesticides and Veterinary Medicine Authority (APVMA) for an emergency use permit to allow for the use of non-registered chlorothalonil formulations. There are a small number of registered chlorothalonil products that have label instructions that do allow for grazing. PIRSA's Rural Chemicals Operations group of Biosecurity SA provided advice for growers regarding the restrictions on grazing instructions when using chlorothalonil applied to pulse crops. Under SA's Agricultural and Veterinary Chemical (Control of Use) legislation, chemical users are required to follow all mandatory instructions and withholding period advice on the label specific to the chemical product and label they are using. For most chlorothalonil products, the label instructions for grazing states 'Do not graze livestock on treated crops'. This advice not to graze livestock must be followed when using a product that has this statement.

Chemical users should be aware that there are a small number of registered chlorothalonil products that have label instructions that do allow for grazing providing the relevant withholding period and export slaughter interval information on the label is followed. These labels state 'Do not graze for 14 days after application', and have additional export slaughter interval statements for livestock going for export that states, 'Livestock that have been grazed or fed treated forage, fodder or stubble should be placed on clean feed for 63 days (nine weeks) prior to export slaughter'. These label statements are the result of the chemical companies that manufactured those specific products providing data when registering the product with the APVMA that supports these grazing claims. Grazing is only allowed when the specific products with label statements that allow for grazing are used. Producers should check with their chemical reseller or consultant which chlorothalonil products allow for grazing.

APVMA pulse permits

Pulse Australia has arranged for the following off-label permits tobe issued by the apvma for use this season (2017). These permits re in addition to those already issued and current.

PER84309	Azoxystrobin / Cyproconozole	Ascochyta in Chickpeas / Lentils
PER84336	Procymidone	Botrytis Grey Mould in Chickpeas
PER84407	Prosaro (TM)	Ascochtya in Chickpeas / Lentils
PER84408	Boscalid	Botrytis Grey Mould in Chickpeas / Lentils
PER84461	Cyprodinil	Ascochtya in Chickpeas / Faba Beans

See the link http://www.pulseaus.com.au/growing-pulses/crop-protection-products.

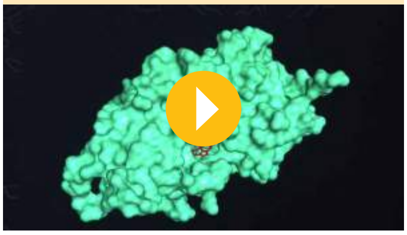
Botrytis grey mould

Botrytis grey mould (BGM) in chickpea is caused by the fungus *Botrytis cinerea*. *B. cinerea* is a significant pathogen of pulse crops particularly lentils, ornamental plants grown under glasshouse conditions, and fruit, including grapes, strawberries and apples. Flowers are especially vulnerable to BGM infection. *B. cinerea* does not infect cereals or grasses.

B. cinerea has been recorded on over 138 genera of plants in 70 families. Legumes and asteraceous plants comprise approximately 20% of these records. As well as being a serious pathogen, *B. cinerea* can infect and invade dying and dead plant tissue. This wide host range and saprophytic capacity means inoculum of *B. cinerea* is rarely limiting. If conditions favour infection and disease development, BGM will occur. This makes management of BGM different from chickpea ascochyta, which is more dependent on inoculum, at least in the early phases of an epidemic.

VIDEOS

2. GCTV9: Botrytis in chickpeas.



B. cinerea also causes pre and post-emergent seedling death. This happens when chickpea seed, infected during a BGM outbreak, is used for sowing. Seedling disease does not need the wet conditions that are usually required for infection and spread of BGM later in the crop cycle. [40]

Economic importance

BGM is a serious disease of chickpeas in southern Australia and can cause total crop failure. Discoloured seed may be rejected or heavily discounted when offered for sale. If seed infection levels are >5% then it may be worth grading the seed. Crop losses are worst in wet seasons, particularly when crops develop very dense canopies.

9.5.8 Varietal resistance or tolerance

See Table 3.

9.5.9 Damage caused by disease

It can be seed-borne, attacking the seedling during emergence and causing rot on the upper taproot and collar. Affected areas develop a soft rot and a fluffy grey mould. Significant losses can occur in wet springs in crops with dense canopies. As well as being a serious pathogen, *B. cinerea* can infect and invade dying and dead plant tissue. This wide host range and saprophytic capacity means inoculum of *B. cinerea* is rarely limiting. If conditions favour infection and disease development, BGM will occur.

This makes management of BGM different from chickpea ascochyta, which is more dependent on inoculum, at least in the early phases of an epidemic.

B. cinerea also causes pre and post-emergent seedling death. This happens when chickpea seed, infected during a BGM outbreak, is used for sowing. Seedling disease does not need the wet conditions that are usually required for infection and spread of BGM later in the crop cycle.

9.5.10 Symptoms

The first symptom of BGM infection in a crop is often drooping of the terminal branches. If groups of plants are infected, these may appear as yellow patches in the crop (Photo 6). The diagnostic feature is a grey 'fuzz' which, under high humidity, develops on flowers, pods, stems and on dead leaves and petioles.

40 M Ryley, K Moore, G Cumming, L Jenkins (2015) Chickpea: managing Botrytis grey mould. Australia Pulse, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/botrytis-grey-mould

Photo 6: *If groups of plants are infected, these may appear as yellow patches in the crop.*

Photo: Phil Davies, Pulse Australia

Lesions can develop anywhere along the stem but are usually first found on the lower part of the stems often starting in leaf axils (Photo 7). Infected seeds are usually smaller than normal and are often covered with white to grey fungal growth.

Photo 7: *Lesions are usually first found on the lower part of the stems often starting in leaf axils.*

Source: Pulse Australia

Infected seeds are usually smaller than normal and are often covered with white to grey fungal growth (Photo 8).

Photo 8: *Botrytis grey mould on seed.*

Photo (left): G. Cumming, Pulse Australia and (right): Pulse Australia

When a severely BGM-infected canopy is opened, clouds of spores are evident (avoid inhaling these). During dry weather the 'fuzz' is not obvious, but it develops again when wet weather returns (Photo 9). Small, dark-brown/black resting bodies (sclerotes) of *B. cinerea* may develop on infected dead tissue, and are capable of producing spores on their surface.

Photo 9: *BGM on a chickpea flower.*

Photo: Phil Davies, Source: Pulse Australia

The stem lesions caused by BGM can be confused with those caused by *Sclerotinia sclerotiorum* (at and above ground level) and by *Sclerotinia minor* (at ground level), but neither of these pathogens produce the grey 'fuzz' typical of BGM. Also, sclerotinia lesions tend to remain white, and are covered by a dense cottony fungal growth, in which irregular shaped black sclerotes develop.

In contrast, the sclerotes of *B. cinerea* are more rounded and usually develop after the stems die. They are smaller than the sclerotes of *S. sclerotiorum*, but larger than the angular sclerotes of *S. minor*. [41]

9.5.11 Conditions favouring development

Factors that favour infection and spread of BGM in favourable seasons include:

- early sowing (mid-April to early May) and narrow rows
- frequent overcast, showery weather
- limited supply of effective fungicides
- lack of BGM tolerant/resistant varieties.

41 M Ryley, K Moore, G Cumming, L Jenkins (2015) Chickpea: managing Botrytis grey mould. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/botrytis-grey-mould

High biomass crops and early canopy closure often results in high in-crop humidity and poor penetration of fungicides. If the crop becomes lodged the situation is exacerbated.

Rainy weather not only favours the disease but wet paddocks also limit the spray opportunities for ground rigs.

Following a season where widespread BGM infection has occurred in a district there is often a shortage of disease-free seed for planting and there is a high quantity of infected crop residue across a large area. Both of these factors will increase the disease risk for the following year. Whether BGM becomes a problem the following year will depend on seasonal conditions.

Over 10 million spores can be produced on a single 2 cm long lesion on a chickpea stem. Consequently, *B. cinerea* has the capacity to rapidly develop during conducive weather conditions. The spores can be blown many kilometres, and if deposited on chickpea plants they can remain dormant until conditions favour spore germination.

Free moisture is necessary for germination and infection. Lesions and the grey 'fuzz' are evident 5–7 days after infection under ideal conditions.

B. cinerea is favoured by moderate temperatures (20–25°C) and frequent rainfall events. It does not become a risk until the average daily temperature (ADT) is 15°C or higher. The combination of early canopy closure, prolonged plant wetness and overcast weather results in high relative humidity and rapid leaf death in the canopy, conditions which are ideal for *B. cinere*a.

B. cinerea can survive on and in infected seeds, in infected stubble, on alternative hosts, in dead plant tissue and as sclerotes. The relative importance of these in Australia is unknown, but recent research in Victoria demonstrated that *B. cinerea* can survive for up to 18 months on infected stubble under field conditions. Other research from WA suggests that sclerotes of *B. cinerea* cannot survive over summer because they lose their viability during hot weather. [42]

Higher seeding rates lead to greater canopy vigour, increased lodging and under ideal growing conditions can increase the risk of BGM.

The fungus survives on infected seed, as a saprophyte on decaying plant debris and as soil-borne sclerotia. The disease is often established in new areas by sowing infected seeds. Masses of spores are produced on infected plants. These fungal spores can be carried from plant to plant by air currents and spread the disease rapidly, (Figure 7). Once a crop has become established, the warm, humid conditions under the crop canopy provide ideal conditions for infection and spread of the disease.

42 M Ryley, K Moore, G Cumming, L Jenkins (2015) Chickpea: managing Botrytis grey mould. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/botrytis-grey-mould

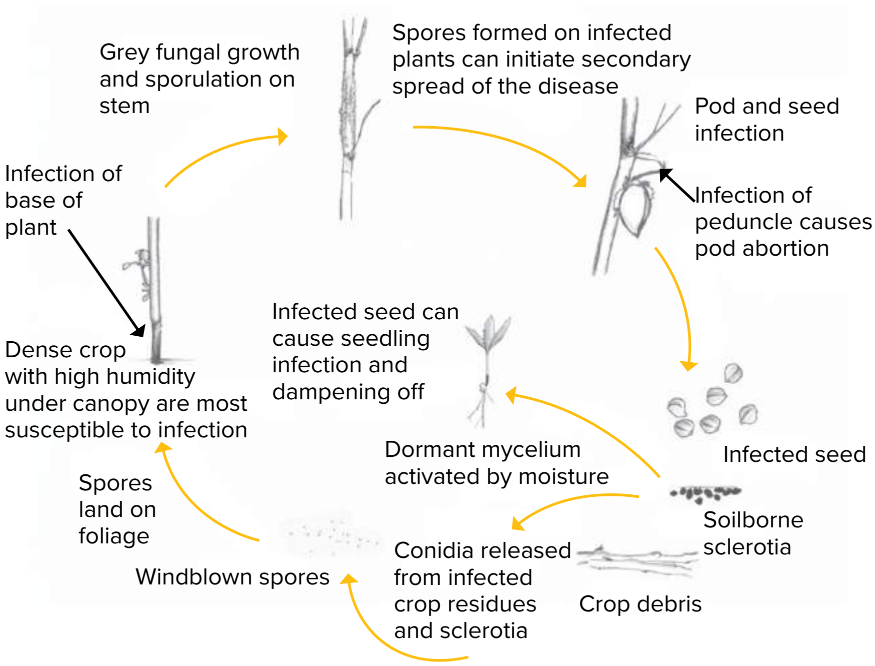


Figure 7: *Disease cycle of Botrytis grey mould in chickpeas.*

Illustration by Kylie Fowler Source: CropPro

9.5.12 Management of disease

Stubble management

It is likely that the pathogen can remain viable and capable of survival for as long as infected stubble remains on the soil surface. Burial of stubble removes the ability of *B. cinerea* to produce spores that can be blown around, and increases the rate of stubble breakdown by soil microbes.

Although burning of infected residues will also significantly reduce the amount of infected residues on the soil surface, it will not guarantee freedom from BGM in the following season.

Burying or burning stubble can significantly increase the risk of soil erosion and reduce water infiltration. [43]

Volunteer control (the green bridge)

Volunteer chickpea plants growing in or near paddocks where BGM was a significant problem are a likely method of carryover and must be managed by application of herbicide or cultivation.

This will also reduce carryover of ascochyta. [44]

Seed source and treatment

Obtain seed from a commercial supplier, or from a source known to have negligible levels of BGM. Irrespective of the source, all seed must be thoroughly treated with a registered fungicide seed dressing. Thiram-based fungicide seed dressings

43 M Ryley, K Moore, G Cumming, L Jenkins (2015) Chickpea: managing Botrytis grey mould. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/botrytis-grey-mould

44 M Ryley, K Moore, G Cumming, L Jenkins (2015) Chickpea: managing Botrytis grey mould. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/botrytis-grey-mould

are effective in significantly reducing, but not entirely eliminating, BGM from infected seed. [45]

See Section 3 Planting, 3.2 Seed treatments for more information.

Seedling emergence

Research on harvested seed has shown a germination test does not accurately predict emergence. Accordingly, growers are advised to conduct their own emergence test, as follows:

- After grading and treatment, sow 100 seeds at least 5 cm deep in the paddock that you intend for chickpeas and water if necessary.
- Count the number of seedlings that have emerged after one, two and three weeks and note their appearance. Do they look healthy or are they stunted and distorted?
- If you want to get an idea of variability in emergence and the paddock, replicate the test i.e. sow 100 seeds in 3–4 different locations in the paddock. This will also help identify potential herbicide residue problems. [46]

Paddock selection

Paddocks in which chickpeas were affected by BGM should not be re-sown to chickpea, faba bean or lentil the following season. Nor should chickpea be sown beside paddocks where BGM was an issue the previous season.

As for Ascochyta blight, chickpeas should be grown as far away from paddocks in which BGM was a problem as is practically possible.

However, under conducive conditions, this practice will not guarantee that crops will remain BGM free, because of the pathogen's wide host range, ability to colonise dead plant tissue, and the airborne nature of its spores. [47]

Sowing time and row spacing

If long-term weather forecasts suggest a wetter-than-normal year (La Nina), consider sowing in the later part of the suggested sowing window for your district and on wider rows (e.g. 100 cm). Planting on wider rows results in increased air movement through the crop and reduced humidity within the canopy. Higher seeding rates lead to greater canopy vigour, increased lodging and under ideal growing conditions can increase the risk of BGM.

Varietal resistance

All current commercial varieties suitable for the northern region are susceptible to BGM, although Howzat⁽ᴰ⁾ is reported to have slightly better resistance than other varieties.

Fungicide treatment

In areas outside central Queensland, spraying for BGM is not needed in most years.

However, in seasons and situations favourable to the disease, a preventative spray of a registered fungicide immediately prior to canopy closure, followed by another application two weeks later, will assist in minimising BGM development in most years. If BGM is detected in a district or in an individual crop, particularly during flowering or pod-fill, a fungicide spray should be applied before the next rain event (Table 10).

45 M Ryley, K Moore, G Cumming, L Jenkins (2015) Chickpea: managing Botrytis grey mould. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/botrytis-grey-mould

46 M Ryley, K Moore, G Cumming, L Jenkins (2015) Chickpea: managing Botrytis grey mould. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/botrytis-grey-mould

47 M Ryley, K Moore, G Cumming, L Jenkins (2015) Chickpea: managing Botrytis grey mould. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/botrytis-grey-mould

None of the fungicides currently registered or under permit for the management of BGM on chickpea have eradicant activity, so their application will not eradicate established infections. Consequently, timely and thorough application is critical. [48]

Sumitomo Sumisclex® 500 fungicide plus other registered products containing: 500 g/L PROCYMIDONE as their only active constituent APVMA Permit PER82976
This permit is in force from 19 July to 30 November 2016 for use in chickpea for the control of Botrytis grey mould.

Table 10: *Foliar fungicides for the control of Ascochyta and Botrytis grey mould.*

Active ingredient	Example trade name	Rate	
		Ascochyta blight	Botrytis grey mould
Chlorothalonil (720 g/L)	Crop Care Barrack® 720# Barrack Betterstick® # Nufarm Unite® 720#	1.0–2.0 L/ha	Not registered
Mancozeb (750 g/kg)	Dithane™ Rainshield™	1.0–2.2 kg/ha	1.0–2.2 kg/ha
Mancozeb (420 g/L)	Penncozeb® SC	1.8–3.95 L/ha	Not registered
Carbendazim (500 g/L)	Spin Flo®	Not registered	500 mL/ha

These are the only registered chlorothalonil products. It is an offence to use any other product. Refer to current product label for complete 'Direction for use' prior to application.

Source: Pulse Australia

9.6 Phytophthora root rot

PRR is a disease of chickpea caused by the fungus-like oomycete *Phytophthora medicaginis*,. It can cause significant yield losses in wetter-than-normal seasons or following periods of soil saturation in normal seasons. Lucerne, perennial and annual medics (*Medicago* species) and other leguminous plants including sulla (*Hedysarum* species) and sesbania (*Sesbania* species) can also host *P. medicaginis*. [49]

PRR is not usually an issue in the southern region.

9.6.1 Varietal resistance or tolerance

See Table 3.

9.6.2 Symptoms

Infection by *P. medicaginis* can occur at any growth stage, causing seed decay, pre- and post-emergence damping-off, loss of lower leaves, and yellowing, wilting and death of older plants (Photo 10). The disease is usually observed late in the season but may also affect young plants. Badly affected seedlings suddenly wither and die with no obvious disease symptoms.

48 M Ryley, K Moore, G Cumming, L Jenkins (2015) Chickpea: managing Botrytis grey mould. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/botrytis-grey-mould

49 K Moore, M Ryley, M Schwinghamer, G Cumming, L Jenkins (2015) Chickpea: managing Phytophthora root rot. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/phytophthora-root-rot

Photo 10: *Cultivated areas killed by phytophthora. Only plants on tops of contours survived (left) and phytophthora in water course (right).*

Photos: Mark Schwinghamer, Source: Pulse Australia

Infected plants are often stunted with obvious yellowing and drying of the foliage. They have few lateral roots and the lower portion of the tap root is often decayed (Photo 11). The remaining tap root is usually discoloured dark-brown to black. Sometimes the discolouration can extend to the base of the plant. The advancing margins of the lesions may also have a reddish-brown discolouration.

Photo 11: *Severely affected plants have no lateral roots (right) and defoliation below tips of stems.*

Photo: Joe Wessels, CropPro

Photo 12: *New roots forming from the top of the taproot.*

Photo: Mike Fuhlbohm, Source: Pulse Australia

Symptoms are sometimes delayed if temperatures are cool and the soil is moist. Lateral roots and tap root die, or dark-brown/black lesions often girdle the taproots (Photo 12). On young plants the lesions may extend up the stem for 10 mm or more above ground level (Photo 13).

Photo 13: *PRR basal lesions extending up the plant stem.*

Photo: Mal Ryley, Source: Pulse Australia

Plants with phytophthora can be easily pulled from the soil. If conditions are mild, affected plants may partially recover by producing new roots from the upper part of the tap root. [50]

9.6.3 Phytophthora and waterlogging

PRR and waterlogging have similar symptoms (Table 11) and are both induced by transient or prolonged soil saturation and surface water. They usually occur in low lying areas of paddocks, or where water accumulates such as on the low side of contour banks or in watercourses, or where the soil has been compacted or has hard pans. However, under very wet conditions, entire paddocks can be affected.

Table 11: *Symptoms of PRR and waterlogging.*

Phytophthora root rot	Waterlogging
Organism kills roots	Low oxygen kills roots
Chickpea, medics, lucerne are hosts	No link with cropping history or weed control
Occurs any time of year	Usually occurs later in the year
Symptoms onset occurs after a week or more	Symptoms onset occurs quite rapidly
Lower leaves often yellow and fall off	Plants die too fast for leaves to yellow or fall
Roots always rotted and discoloured	Initially roots not rotted or discoloured (tips black)
Plants easily pulled up and out	Plants not easily pulled up initially
Manage through paddock rotation varietal choice	Manage through paddock selection, no irrigation in reproductive phase

Source: Pulse Australia

Symptoms of waterlogging can be confused with those of phytophthora but differ in that:

- plants are most susceptible to waterlogging at flowering and early pod-fill
- symptoms develop within two days of flooding compared to at least seven days for phytophthora
- roots are not rotted and are not easily pulled from the soil at first
- plants often die too quickly for the lower leaves to drop off.

9.6.4 Conditions favouring development

Phytophthora medicaginis survives in soil mainly as thick-walled oospores, but some strains also survive as chlamydospores. Oospores can survive in soil for at least 10 years. In saturated soil the exudates from the roots of chickpea and other hosts stimulate the oospores to germinate and produce lemon-shaped sporangia. Inside these sporangia, zoospores develop and are released into the soil and surface water, where they are carried by moving water and 'swim' towards the roots and collars of chickpea plants.

Zoospores encyst on the root surfaces and germinate to produce hyphae that invade the roots. New sporangia develop from infected roots enabling further cycles of infection to occur. Later, oospores are formed in the infected roots.

Zoospores are only capable of 'swimming' for a few millimetres, so long distance dispersal of *P. medicaginis* is by physical movement of soil and water infested with

50 K Moore, M Ryley, M Schwinghamer, G Cumming, L Jenkins (2015) Chickpea: managing Phytophthora root rot. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/phytophthora-root-rot

oospores, sporangia, zoospores and/or chlamydospores during floods and irrigation or by machinery. [51]

9.6.5 Management of PRR

Though, PRR is not usually an issue in the southern region, the disease can be reduced by seed treatments containing metalaxyl (Apron XL®, Rampart® or Mantle®). [52]

Once a plant or crop is infected with phytophthora, there is nothing a grower can do.

There are no effective chemical sprays as there are for ascochyta and botrytis. Therefore, phytophthora can only be managed by pre-sowing decisions and assessing risks for individual paddocks.

Development of the disease requires both the pathogen in the soil, and a period of soil saturation with water. Losses in a phytophthora-infested paddock may be minor if soil saturation does not occur.

The most effective control strategy is to not sow chickpeas in high-risk paddocks, which are those with a history of:

- phytophthora noted in previous chickpea or lucerne crops
- lucerne or annual or perennial medics
- waterlogging or prone to flooding
- metalaxyl-based seed dressings
- poor drainage.

Do not flood irrigate after podding has commenced especially if the crop has been stressed.

However, if you choose to sow chickpeas in high-risk paddocks, the following measure will reduce losses from phytophthora:

- Growing a chickpea variety with the highest level of resistance. Particularly in medium-risk situations, where medic, chickpea or lucerne crops have been grown in the past 5–6 years. [53]

9.6.6 New tool to determine risk of chickpea PRR

Key points:

- Increasing level of inoculum (oospores/plant) of *Phytophthora medicaginis* (Pm) was strongly correlated with decreasing yield of the moderately resistant variety YORKER⊕.
- An inoculum level of 660 oospores/plant (PreDicta B™ > 5000 Pm copies/g soil) at sowing significantly reduced yields compared with lower inoculum levels under both dryland and irrigated conditions.
- Testing soil samples from growers' 2015 paddocks confirmed the results of testing 2014 samples that the PreDicta B™ soil Pm test can identify Pm in growers' paddocks.
- These findings provide further evidence that the PreDicta B™ Pm test will be a useful tool for growers to determine their risk of PRR.

51 K Moore, M Ryley, M Schwinghamer, G Cumming, L Jenkins (2015) Chickpea: managing Phytophthora root rot. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/phytophthora-root-rot

52 W Hawthorne, J Davidson, L McMurray, K Lindbeck, J Brand (2012) Chickpea disease management strategy—Southern region. Pulse Australia, Southern Pulse Bulletin PA 2012 #08, http://pulseaus.com.au/storage/app/media/crops/2012_SPB-Chickpea-disease-management.pdf

53 K Moore, M Ryley, M Schwinghamer, G Cumming, L Jenkins (2015) Chickpea: managing Phytophthora root rot. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/phytophthora-root-rot

Phytophthora medicaginis detection in soil

Phytophthora medicaginis (Pm), the cause of chickpea PRR) is endemic and widespread in northern to central NSW. Under conducive conditions, PRR can cause 100% loss. The pathogen survives from season to season on chickpea volunteers, lucerne, native medics, sulla and as resistant structures (oospores) in roots and soil.

A PreDicta B™ soil DNA test has been developed by the South Australian Research and Development Institute (SARDI) to quantify the amount of Pm DNA in soil samples and so provide a measure of the amount of Pm inoculum (infected root tissue and oospores) in paddocks. SARDI reports on the second season of studies to assess the capability of this test to:

1. Detect Pm inoculum in soil from commercial paddocks
2. Predict the risk of PRR disease and potential yield losses in chickpea

Pm DNA sampling in paddocks and disease risk determination

The DNA result for a soil sample from a paddock can only provide an indication of inoculum concentration and disease risk for the areas of the paddock which were sampled. Therefore, the spread and locations of sampling across a paddock will affect how representative DNA results are for a paddock. Because of the risk of rapid PRR disease build-up following wet conditions it may be appropriate to treat a negative PreDicta B™ test result as indicating a low risk rather than a nil risk, as the pathogen could still be in areas of the paddock that were not sampled and so still cause PRR and reduce yield.

To maximise the probability of determining the PRR risk of a paddock, target those areas of the paddock where Pm is more likely to occur. The pathogen thrives in high soil moisture contents and so often occurs in low lying regions of paddocks where pooling following rain may occur. The pathogen also carries over from season to season on infected chickpea volunteers, lucerne and, native medics. Including low lying areas and weedy areas of paddocks during PreDicta B™ soil sampling may provide the best strategy to identifying a paddocks disease risk of PRR in chickpea. [54]

MORE INFORMATION

GRDC Update paper; A new DNA tool to determine risk of chickpea Phytophthora root rot.

9.7 Sclerotinia

Sclerotinia, caused by *Sclerotinia sclerotiorum* and *trifoliorum*, is an occasional disease of chickpeas but has caused significant crop losses in eastern Australia. Sclerotinia can cause serious crop losses where a substantial number of plants within a crop are affected. Kabuli chickpeas appear more susceptible to this disease than desi chickpeas but both types can be seriously damaged under favourable conditions. Dense crops are likely to be the most severely affected, particularly under moist conditions. Grain quality can be decreased when infected with sclerotinia, which causes poor colour and shrivelled seed. [55]

9.7.1 Damage caused by disease

Sclerotinia can cause severe damage in chickpeas. This has occurred in kabuli chickpeas in Victoria. Sclerotinia has caused significant crop losses where a substantial amount of the crop is infected. This disease has caused total crop failure where chickpeas were sown in the same paddock in successive years. However, in many situations it only affects a small proportion of plants within the crop.

Kabuli chickpeas are most susceptible to this disease though desi chickpea can also be badly affected under conditions favourable for the disease. Dense crops are likely

54 S Bithell, K Moore, K Hobson, S Haden, W Martin, A McKay (2016) A new DNA tool to determine risk of chickpea Phytophthora root rot. GRDC Update Papers 01 March 2016, https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/03/A-new-DNA-tool-to-determine-risk-of-chickpea-Phytophthora-root-rot

55 CropPro. Sclerotinia of chickpeas. Identification and management of field crop diseases in Victoria, Diseases in pulses: chickpeas, http://www.croppro.com.au/crop_disease_manual/ch05s03.php

to be affected, particularly under moist conditions. Grain quality can be decreased when infected with sclerotinia. It causes poor colour and shrivelled seed. [56]

9.7.2 Symptoms

There are two *Sclerotinia* spp that attack chickpeas and they can be distinguished by the size of their sclerotes (survival structures):

- *S. sclerotiorum* produces large irregular shaped sclerotes 5–10 mm in diameter as high up as 20–30 cm on the stem
- *S. minor* produces sclerotes that are angular and much smaller, rarely larger than 2–3 mm in diameter

What to look for:

A small number of dead plants scattered throughout a paddock. Affected plants first wilt and rapidly die, often without turning yellow. Later, as the plant dries out the leaves turn a straw colour (Photo 14).

Photo 14: *Plants killed by S. sclerotiorum (left). Fungal weft of sclerotinia in the lower canopy of a chickpea crop (right).*

Photos: Kevin Moore, Source: Pulse Australia

On the surface of the root, just below ground level, small black fungal bodies called sclerotia (which are irregular in size and shape), can sometimes be seen mingled with white cottony fungal mycelium.

In spring, water-soaked spots may appear on the stems and leaves. Affected tissues develop a slimy soft rot from which droplets of a brown liquid may exude. Infected tissues then dry out and may become covered with a web of white mycelium of the fungus (Photos 15 and 16).

Photo 15: *Early symptoms of stem infection by sclerotinia. White mycelial growth starting to develop (left). RIGHT: Comparison of stem infections caused by sclerotinia (top stem) and botrytis (lower stem)—note the different colour of fungal growth.*

Source: CropPro

56 AgVic (2008) Sclerotinia of chickpea. Agriculture Victoria, Ag Note AG0453 June 2008 Updated August 2012, http://agriculture.vic.gov.au/agriculture/pests-diseases-and-weeds/plant-diseases/grains-pulses-and-cereals/sclerotinia-of-chickpea

Photo 16: *Sclerotinia stem infection of chickpeas. White fluffy mycelium and sclerotia formation evident.*

Source: CropPro

9.7.3 Conditions favouring development

The disease is usually established from sclerotia (survival bodies of the fungus) present in the soil or introduced with contaminated seed. Outbreaks are more common when very wet conditions occur in July.

The sclerotia germinate in moist soil and either directly infect roots or produce airborne spores (Photo 17) which attack the above-ground parts of the plant. Once established, the fungus rapidly moves to adjacent healthy tissue. Within a few days of infection, plants start to wither then die.

Photo 17: *Ascospore infection of chickpea stem by S. sclerotiorum.*

Photo: G Cumming, Pulse Australia

Sclerotia formed on infected plants enable the fungus to survive to the following year. Individual seeds can be contaminated with the fungus and/or sclerotia may be present in the seed sample. Sclerotia can remain viable in the soil for up to eight years (Figure 8).

Soil-borne sclerotia are the most important disease source for establishing disease in following crops. Seeds infected with sclerotinia can be the cause of disease establishment in otherwise sclerotinia-free areas. [57]

57 CropPro. Sclerotinia of chickpeas. Identification and management of field crop diseases in Victoria, Diseases in pulses: chickpeas, http://www.croppro.com.au/crop_disease_manual/ch05s03.php

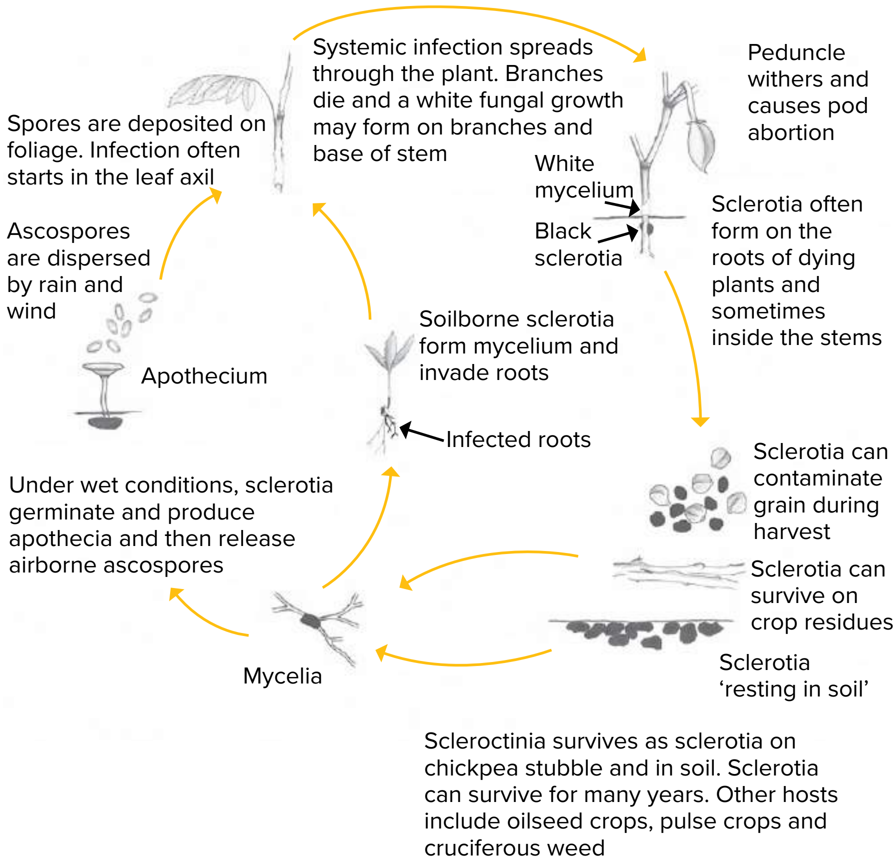


Figure 8: *Disease cycle of sclerotinia of chickpea.*

Illustration by Kylie Fowler, CropPro

9.7.4 Management of sclerotinia

Before sowing

Use clean seed

Use of disease-free seed minimises the risk of disease and prevents establishment into a new area. It is important to avoid sowing chickpea in areas where the disease is known to be present. The seed harvested from infected crops should not be used for sowing.

Crop rotation

Crop rotation is the best method of control once the disease has become established. Cereal crops are not affected by sclerotinia and provide a good disease break. Pulse crops, oilseeds, legume-based pastures and capeweed are all good hosts to this disease.

If a severe sclerotinia problem does occur, a four-year break from susceptible crops is required to substantially reduce the number of sclerotia in the soil. The most practical option is to use cereals and legumes such as field peas or vetch which have some resistance to sclerotinia. In addition, burning of the disease-infected stubble should be considered. Deep ploughing (5 cm) will also reduce the number of sclerotia, and so minimise disease carry over. Where a minor sclerotinia problem occurs, a two-year break from susceptible crops is advisable.

No commercial seed treatments or fungicides are known to manage this disease in crop. [58]

IN FOCUS

Use of non-conventional chemicals as an alternative approach to protect against sclerotinia

Four non-conventional chemicals, viz., zinc sulphate (ZS), oxalic acid (OA), sodium malonate (SM) and sodium selenite (SS), were applied as foliar sprays to chickpea and the plants were subsequently challenged against *Sclerotinia sclerotiorum,* the causal agent of stem rot in chickpea. All the chemicals reduced mortality of chickpea from *S. sclerotiorum* infection. Among them, ZS at 10-3 mmol gave the best result as only 13.6% mortality was recorded after 28 days compared to 100% in the control. High performance liquid chromatographic analysis of treated chickpea leaves revealed activation of shikimic acid as well as phenyl propanoid pathways and synthesis of several phenolic compounds increased specially after application of OA, ZS and SM. Individual treatment of the chemicals showed better results than their combinations as plant mortality was reduced and accumulation of phenolics increased in their individual treatments. A positive correlation was observed between induction of phenolic compounds and survival of the plants. In vitro assay of the four chemicals showed only SS to be antifungal. The protection of plants by ZS, OA and SM is possibly because of induction of resistance in the host against *S. sclerotiorum.* [59]

Consult your local agronomist about potential treatment strategies for your area.

9.8 Phoma stem rot

Phoma caused by the fungus *Phoma medicaginis* var. *pinodella*, has the potential to be a serious disease of chickpea. Relatively few serious outbreaks have occurred; however, the disease is common in southern Australia. The disease can cause serious crop losses in seasons with above average winter rainfall. Careful paddock selection and use of fungicide seed dressings can minimise the impact of this disease.

9.8.1 Symptoms

What to look for

Seed-borne infection often results in black-brown discolouration of the root near where the seed is attached (Photo 18). Blackening may spread up the root and cause lesions at the base of the stem.

58 AgVic (2008) Sclerotinia of chickpea. Agriculture Victoria, Ag Note AG0453 June 2008 Updated August 2012, http://agriculture.vic.gov.au/agriculture/pests-diseases-and-weeds/plant-diseases/grains-pulses-and-cereals/sclerotinia-of-chickpea

59 BK Sarma, SA Basha, DP Singh, UP Singh (2007) Use of non-conventional chemicals as an alternative approach to protect chickpea (*Cicer arietinum*) from Sclerotinia stem rot. Crop Protection, 26(7), 1042-1048.

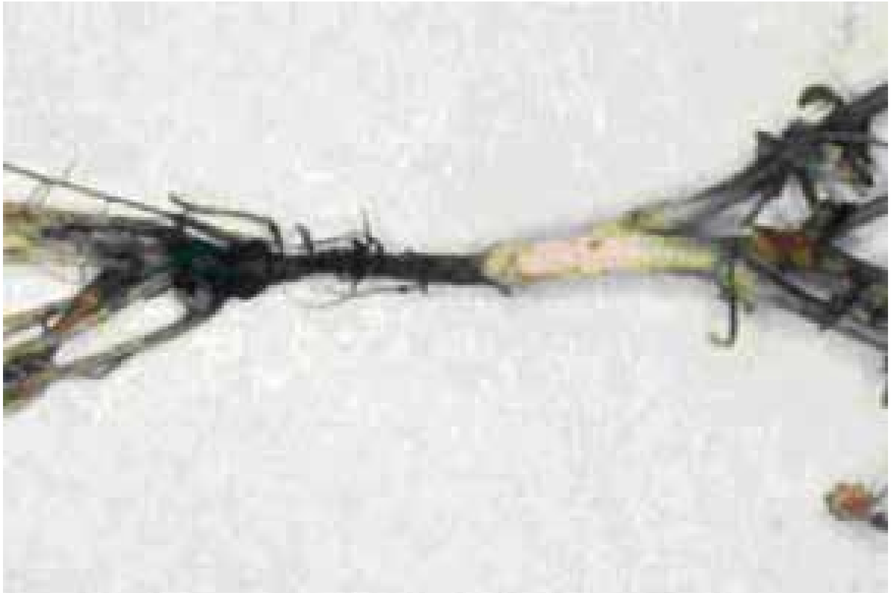


Photo 18: *Characteristic collar and root rot caused by Phoma.*

Source: CropPro

Initial above-ground symptoms are small, dark tan coloured, irregular flecks on leaves, stems, and pods. The flecks on leaves enlarge to lesions and the surrounding tissue yellows. Within the lesions numerous pinhead-sized black fruiting bodies of the fungus develop. On the stem, similar but more elongated lesions form.

Black lesions may completely girdle the base of the stem and root where infection is severe (Photo 19). Pod lesions are sunken, with pale centres and dark margins, and may be covered by small black spots. The fungus may penetrate the pod and infect developing seeds. Badly affected plants may be totally defoliated when infected leaflets senesce and fall.

Photo 19: *Stem lesions caused by Phoma infection.*

Source: CropPro

9.8.2 Conditions favouring development

Phoma can survive on infected seed, in soil and on crop residue from one season to the next. Infection can occur at any stage of plant growth provided conditions are favourable. Moisture is essential for infection to occur. During wet weather, the disease may spread further when spores of the fungus are carried by wind and rain splash onto neighbouring plants (Figure 9). Pod infection can occur when the fungus penetrates the pod wall and infects developing seeds late in the season.

The only serious outbreaks of this disease on chickpea in Australia have occurred in very wet years. However, it is usually a more damaging disease on field pea than chickpea.

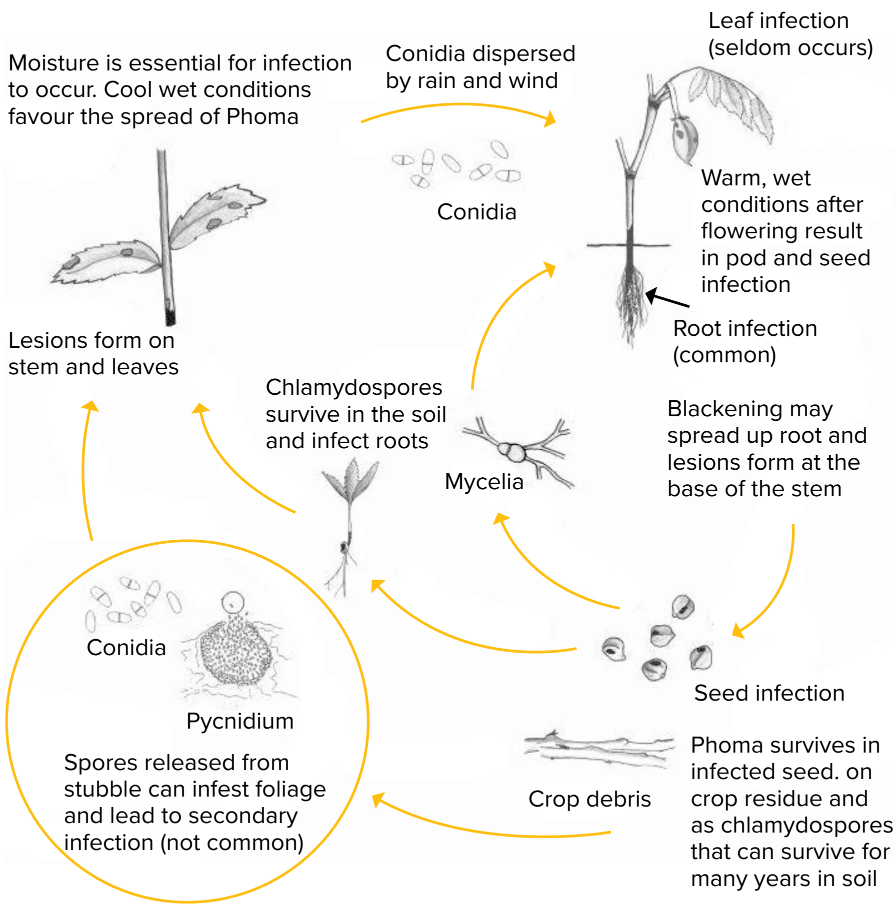


Figure 9: *Disease cycle of Phoma stem rot of chickpeas.*

Illustration by Kylie Fowler, CropPro

9.8.3 Managing Phoma

Before sowing

Use clean seed

The use of disease-free seed and crop rotation will help prevent the establishment and build-up of this disease.

Crop rotation

Where chickpeas have been badly infected, a two-year break from host crops will minimise the disease risk. Crops which host Phoma include field pea, chickpea, faba bean, lupin, lentil, vetch and legume pasture species. Cereal and oilseed crops will provide a good disease break.

Chemical control

Seed-borne disease infection can sometimes be controlled with fungicide seed dressings. No fungicides are known to manage this disease in crop. [60]

9.9 Root rots including damping-off (*Fusarium, Rhizoctonia* and *Pythium* spp.)

All fungi responsible for root rot are soil dwellers. They can survive from crop to crop in the soil, either on infected plant debris or as resting spores. In wet soils, these fungi can invade plant roots and cause root rot. Wet conditions also encourage the spread of disease within a field.

9.9.1 Economic importance

Root rot diseases can occasionally be serious especially when soils are wet for prolonged periods. The reduced root development causes the plants to die when they are stressed.

9.9.2 Symptoms

Affected seedlings gradually turn yellow and leaves droop. The plants usually do not collapse. The taproot may become quite brittle, except in Pythium root rot when they become soft. When plants are pulled from the ground the portion of the root snaps off and remains in the soil. The upper portion of the taproot is dark, shows signs of rotting and may lack lateral roots. Distinct dark-brown to black lesions may be visible on the taproot (Photo 20). The leaves and stems of affected plants are usually straw-coloured, but in some cases may turn brown. Older plants dry-off prematurely and are often seen scattered across a field. In some cases, especially with kabuli, seeds may rot before they emerge.

60 AgVic (2008) Phoma in chickpea. Agriculture Victoria, Ag Note AG0452 June 2008 Updated August 2012, http://agriculture.vic.gov.au/agriculture/pests-diseases-and-weeds/plant-diseases/grains-pulses-and-cereals/phoma-of-chickpea

Photo 20: *Rhizoctonia root rot. Optimum soil temperature is 24–26°C; disease is worse on light sandy soils.*

9.9.3 Management options

Root rot disease can be reduced by crop rotation. As this disease may also affect other pulses, chickpeas should be sown in rotation with another non-legume crop. Chickpeas should not be grown in areas subject to waterlogging. Damping-off in kabuli chickpeas can be controlled using fungicide seed treatment.

9.10 Collar rot (*Sclerotium rolfsii*)

9.10.1 Economic importance

Collar rot is generally a minor disease in chickpea. However, the disease has been particularly severe in irrigated Macarena (kabuli).

9.10.2 Symptoms

This disease is commonly observed at very low levels in chickpea crops (up to 6 weeks after sowing) sown during warmer conditions, as isolated dead seedlings with a coarse web of white fungal threads encasing the tap root. However, in irrigated systems, the fungus can kill significant numbers of plants. The coarse threads of the fungus can be seen on or just under the soil surface, colonising decomposing trash or on the plant itself (Photo 21); these webs of mycelium can cover quite a substantial area around plants. On chickpea, plants will be killed outright and quite rapidly as the fungus invades around the soil level and girdles the vascular tissue. Plants will wilt and become bleached (a result of a toxin produced by the fungus), younger seedlings may collapse but older plants may simply dry (without collapse). The characteristic signs of the pathogen will be the webs of coarse mycelium and the small (~1–2 mm) spherical brown sclerotia (survival and resting structures) of the fungus that attach to the fungal threads. The sclerotia look like canola seeds.

Photo 21: *Webs of Sclerotium rolfsii mycelium at the base of an infected chickpea plant.*

Photo: K McCosker

9.10.3 Conditions favouring development

The fungus has a very wide host range including monocots (such as millet and barley) and dicots (such as cotton). The pathogen is also the causal agent of white mould in peanuts.

The pathogen rarely occurs where average winter temperatures fall below 0°C. The fungus survives in the soil mainly as sclerotia that remain viable for 2–3 years, but occasionally it persists as mycelium in infected tissues or plant residues. Sclerotia germinate by hyphal or eruptive germination. Hyphal germination is characterised by the growth of individual hyphae from the sclerotial surface, while eruptive germination is characterised by plugs or aggregates of mycelium bursting through the sclerotial surface. [61]

9.10.4 Management options

The disease is favoured by the presence of undecomposed organic matter on the soil surface and excessive moisture. If possible, avoid wetting and drying cycles during warmer periods, as this promotes germination of the sclerotia, and try to minimise inter-row cultivation, which pushes soil up around the base of plants. The fungus is a very effective saprophyte of cotton trash, so allowing time for cotton trash to break down prior to planting will reduce the activity of the fungus. Similarly, trash from other crops such as barley and millet are attractive substrates for the fungus.

9.11 Fungal disease control

9.11.1 When to spray

Sprays will control fungal disease, but when and how often to spray will depend on the varietal resistance, amount of infection, the impending weather conditions and the potential yield of the pulse crop.

61 ZK Punja (1985) The biology, ecology, and control of *Sclerotium rolfii*. Annual review of Phytopathology, 23(1), 97-127.

Fungal disease control is geared around protection rather than cure. The first fungicide spray must be applied as early as necessary to minimise the spread of the disease. Additional sprays are required if the weather conditions favour the disease.

9.11.2 Principles of spraying

A fungicide spray at the commencement of flowering protects early pod-set. Additional protection may be needed in longer growing seasons until the end of flowering. Fungicides last around 2–3 weeks.

Remember all new growth after spraying is unprotected. Coverage and canopy penetration is critical, as only treated foliage will be protected. Translocation is very low in most products.

In periods of rapid growth and intense rain (50 mm over several days), the protection period will reduce to ~10 days.

Timing of fungicide sprays is critical (Table 12 and 13). As Ascochyta blight and BGM can spread rapidly, DO NOT DELAY spraying. A spray in advance of a rainy period is most desirable.

Despite some fungicide washing off, the disease will be controlled. Delaying until after a rainy period will decrease the effectiveness of the fungicide as the disease has started to spread.

Repeat fungicide sprays depend on:

- amount of unprotected growth
- rainfall since spraying
- likelihood of a further extended rainy period.

Unprotected crops can lose >50% in yield. In severe cases, the crop may drop all of its leaves. [62]

62 Pulse Australia (2016), Chickpea fungicide guide: 2016 season. Pulse Australia, Australian Pulse Bulletin, http://pulseaus.com.au/growing-pulses/bmp/chickpea/2016-season-fungicide-guide

Table 12: *Principles of when to spray for fungal disease control in chickpea.* [63]

Disease	Occurrence	When to spray
Ascochyta blight	First appears under wet conditions	Resistant variety. Fungicide sprays are unlikely to be required before podding. Despite good foliar resistance to Ascochyta blight, the flowers and pods of resistant varieties can be infected which can result in poor quality, discoloured seed or seed abortion and, in extreme situations, yield loss. Moderately resistant variety. In most seasons, disease development will be slow and there will be no or minimal yield loss. In such seasons there is no cost benefit in applying a fungicide during the vegetative stage. Despite good foliar resistance to Ascochyta blight, the flowers and pods of MR/R rated varieties can be infected, which can result in poor quality, discoloured seed or seed abortion and yield loss in severe situations. However, under high disease pressure, a reactive foliar fungicide strategy may be warranted during the vegetative period of the crop. If Ascochyta blight is present in the crop, apply a registered fungicide at early podding prior to rain to ensure pods are protected, and high quality, disease-free seed is produced. Susceptible variety. If the season favours Ascochyta blight, regular fungicide sprays will be needed from emergence until 4 weeks before maturity. Do not wait until you find the disease. Timing of the first two sprays is critical, because control is difficult or impossible after the disease has taken hold. The first spray must be applied before the first post-emergent rain event, or 3 weeks after emergence or at the 3-leaf stage, whichever occurs first. The second spray should be applied 3 weeks after the first spray. However, apply the second spray if 2 weeks have elapsed since the first spray and rain is forecast. Continue to monitor the crop 10–14 days after each rain event. If Ascochyta blight is found, additional sprays will be required. If it has been ≥2 weeks since the last application, spray again just before the next rain event. For all varieties regardless of resistance. If Ascochyta blight is detected, apply a registered fungicide at early podding prior to rain. In high-rainfall or high-risk situations and where there is an extended pod-filling period, further applications may be required
Botrytis grey mould	Develops during warm (15–20°C), humid (>70%) conditions, usually at flowering	During early to mid-flowering as a protective spray. Additional sprays may be necessary through flowering and pod-filling if disease progresses. Disease is favoured by warm weather (15–20°C) and high humidity (>70% RH)

63 K Moore, M Ryley, G Cumming, L Jenkins. (2015) Chickpea: Ascochyta blight management. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/ascochyta-blight

Table 13: *Carryover of major pulse diseases, showing relative importance as sources of infection.* [64]

Disease	Stubble	Seed	Soil
Ascochyta blight	***	**	*
Botrytis grey mould	***	***	*
Phytophthora root rot			***
Sclerotinia	*	*	***

9.12 Viruses

Key points:

- Chickpea is distinct from other pulses in respect to virus diseases and how viruses spread in crops.
- Aphicide sprays and some other control strategies that are effective in other pulses are less effective.
- At present, the best control options for chickpea are the current best agronomic practices: retaining standing stubble, using optimal sowing rates and times, and controlling in-crop and fallow weeds. [65]
- Virus management aims at prevention through integrated management practice that involves controlling the virus source, aphid populations and virus transmission into pulse crops.
- Rotate legume crops with cereals to reduce virus and vector sources and where possible avoid close proximity to perennial pastures (e.g. lucerne) or other crops that host viruses and aphid vectors.
- Eliminate summer weeds and self-sown pulses 'green bridge' that are a host for viruses and a refuge for aphids.
- Aphid activity is influenced by seasonal conditions and will require early monitoring in nearby crops and pastures and possible use of an aphicide or cultural controls to reduce numbers.
- Sow directly into cereal stubbles (preferably standing), and encourage rapid canopy cover through early planting, high planting density as bare soil is more attractive to some aphid species.
- Purchase virus-tested seed or have farmer seed virus tested as PSbMV, CMV, BYMV and AMV depend largely on seed transmissions for survival.
- Gaucho® 350SD is now registered and when applied as seed treatment will help protect faba bean, field pea and lentil seedlings from early season aphid attack and reduce virus spread.

Viruses differ from most fungal diseases in that they infect plants systematically and no curative treatment is available. Virus infections are spasmodic and levels depend heavily on seasonal conditions and differ greatly between years and locations. Early infection can lead to stunting, reduced tillering and plant death and losses can be high. Late infections have less impact, but can still affect seed quality. [66]

There are more than 14 species of virus that naturally infect chickpeas. These viruses are spread by airborne insects, with aphids being the predominant vector. The occurrence of virus in chickpeas is episodic and changes dramatically from season to season and location. Clovers, medics, canola/mustard, weeds and other pulses can host viruses that infect chickpea. The best control strategies to reduce risk of viruses are agronomic. These include retaining cereal stubble, sowing on time, establishing a

64 Pulse Australia, Checklist for Northern growers http://www.pulseaus.com.au/storage/app/media/crops/2010_NPB-Chickpea-checklist-north.pdf

65 M Schwinghamer, T Knights, K Moore (2009) Virus control in chickpea—special considerations. Pulse Australia, Australian Pulse Bulletin PA 2009 #10, http://www.pulseaus.com.au/storage/app/media/crops/2009_APB-Chickpea-virus-contol.pdf

66 Pulse Australia (2015) Managing viruses in pulses. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/publications/manage-viruses

uniform closed canopy and controlling weeds. [67] Seed and foliar insecticides are not recommended for chickpea viruses. [68]

9.12.1 Symptoms

Viruses exhibit a varied range of symptoms and severity from relatively unapparent to plant death. The intensity and symptoms depend on virus and pulse species and to a lesser extent on virus strain, pulse variety, climatic conditions and plant stage at infection. Plants infected at an early stage or through seed will usually show more uniform discolouration and stunting, but when infected at the later stage will usually occur at the leaf tip before the whole plant starts to deteriorate (Photo 22).

Photo 22: *Kabuli chickpea (centre) with low plant stand and high virus infection compared to kabuli (right) and desi (left) with good canopy.*

Source: Pulse Australia

Foliage symptoms are often more visible on young leaves and can include yellowing (sometimes reddening), vein clearing, leaf mottle, leaf distortion, curling of leaves, reduced size, chlorotic or necrotic spotting, or more widespread necrosis (Photo 23). Shoot symptoms may be seen as bunching of young leaves, growth of auxiliary shoots, bending over of the growing point, tip or apical necrosis, streaking of stems, stunting and wilting or plant death.

67 Schwinghamer *et al.* 2009, Verrell 2013, Murray *et al.* 2014

68 A Verrell (2013) Virus in chickpea in northern NSW 2012. GRDC Update Papers. 26 February 2013, http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/02/Virus-in-chickpea-in-northern-NSW-2012

Photo 23: *Wilting and necrosis of shoot tip in desi chickpea cv. Amethyst infected naturally with Tomato spotted wilt virus (TSWV).* [69]

Symptoms such as leaf yellowing, veining, mottling, and wilting can often be confused with nutrient deficiencies, herbicide damage or water stress unless sufficiently distinct. It is also difficult to tell which virus is present without resorting to laboratory tests on plant samples.

It is best to collect living tissue samples and collection and packaging of fresh samples is simple. Instructions from local agronomists or Pulse Australia need to be heeded. Immediately place the sample with paper towelling into a plastic bag, seal it and refrigerate it until dispatched. Send the sample by priority post and do not leave it sitting around. [70]

9.12.2 Conditions favouring development

High levels of virus infections have occurred in recent years resulting from infected plants in the previous spring as a virus source and a 'green bridge' of summer plant material to carry over these viruses and as a refuge for aphids. Warm dry conditions during autumn have favoured increased aphid activity and virus transmission.

Some aphid species prefer to land on plants surrounded by bare ground and favour thin crop stands or areas within the crop which have low plant densities.

Stressed plants are also more attractive to aphids, possibly due to a higher level of plant sugars, and are vulnerable to colonisation and can become a source of virus spread. Environmental factors that impacted on chickpeas in 2009 were extremely dry conditions early in the season that favoured aphid build and this was particularly evident in vetch crops. Then followed cold and wet conditions that included some transient waterlogging that stressed plants making them more venerable to root diseases and aphid attack.

Chickpea that border lentil, canola or lucerne crops can be subjected to larger numbers of aphids, as they can readily colonise these crops and multiply quickly.

69 JE Thomas, MW Schwinghamer, JN Parry, M Sharman, MA Schilg, EK Dann (2004). First report of Tomato spotted wilt virus in chickpea (*Cicer arietinum*) in Australia. Australasian Plant Pathology, 33(4), 597-599, http://era.daf.qld.gov.au/634/1/Thomas_tomato.pdf

70 Pulse Australia (2015) Managing viruses in pulses. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/publications/manage-viruses

Controlling aphids in these nearby host crops can potentially decrease aphid numbers moving through chickpea crops.

Types of transmission

Pulse viruses are transmitted either in a persistent or non-persistent manner by insects (mostly aphids). The mode of transmission has implications for the way a virus develops in the field and its management.

Persistently transmitted viruses:

- Bean leafroll virus (BLRV)
- Beet western yellows virus (BWYV) (Photo 24)
- Subterranean clover red leaf virus (SCRLV)
- Subterranean clover stunt virus (SCSV) [71]

Photo 24: *Beet Western Yellows Virus in kabuli (top) and in desi (bottom).*

Source: CropPro.

The general symptoms of BLRV on pulses are interveinal chlorosis, yellowing, stunting and leaf rolling (Photo 25). These symptoms could easily be confused with subterranean clover stunt virus (SCSV) or other luteoviruses such as beet western yellows virus (BWYV) and subterranean clover red leaf virus (SCRLV) or nutrient stress symptoms. [72]

71 Pulse Australia (2015) Managing viruses in pulses. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/publications/manage-viruses

72 Agriculture Victoria. (2013). Temperate pulse viruses: Bean leafroll Virus. http://agriculture.vic.gov.au/agriculture/pests-diseases-and-weeds/plant-diseases/grains-pulses-and-cereals/temperate-pulse-viruses-bean-leafroll-virus-blrv

Photo 25: *Chickpeas develop tip yellowing and stunting.*

Photo: S Kumari, ICARDA, Source: AgVic.

Persistent transmission means that once the insect becomes infectious, it remains so for the rest of its life. After an insect vector feeds on an infected plant, the virus has to pass through its body and lodge in the salivary glands before it can be transmitted to healthy plants. Not all aphid species are vectors of this kind of virus in pulses so the identification of aphid species is very important.

BWYV is the main virus and most common occurring in chickpea and lentil crops. It has a diverse natural host range including canola, pasture plants, lucerne and many weeds such as paddy melons, wild radish and some native legumes. BLRV is another but is limited to fabaceae (faba bean, field pea, chickpea, and lentil), lucerne, clovers and summer legumes.

Persistently transmitted viruses typically start with a random distribution of infected plants in autumn and increase during the season as vectors colonise the crop. Transmission rates can dramatically increase with large aphid flights that will often coincide with aphid activity and build-up prior to sowing.

Non-persistently transmitted viruses:

- Alfalfa mosaic virus (AMV)
- Bean yellow mosaic virus (BYMV)
- Cucumber mosaic virus (CMV)
- Pea seed-borne mosaic virus (PSbMV)

Non-persistently transmitted viruses can be seed-borne (depending on the virus/crop combination), but require aphid vectors to spread during the season. [73]

9.12.3 Reducing risk of viral diseases

Controlling virus disease in chickpeas is difficult. Chickpea plants that become infected with a virus invariably die. GRDC-funded field trials have shown no benefit of seed-applied insecticides or regular foliar-applied insecticides or a combination of both against chickpea viruses. The best and at this stage only, control strategies to reduce risk of viruses in chickpeas are agronomic. These include; retaining cereal stubble, sowing on time, establishing a uniform closed canopy, providing adequate nutrition and controlling weeds. Reduce risk of viruses in chickpea crops by planting

73 Pulse Australia (2015) Managing viruses in pulses. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/publications/manage-viruses

between rows of standing cereal stubble, sowing on time and targeting at least 25 plants/m^2. [74]

9.12.4 Management of viruses

A virus management strategy to reduce the risk of infection may require a number of control measures relevant to the various virus and pulse types.

Better agronomy – better chickpeas

Field trials from 2012 and 2013 have shown that chickpea crops are at risk of increased damage from viruses when plant density is <20 plants/m^2. Significantly fewer plants are infected when plant densities are higher, and it is recommended to aim for >25 plants/m^2.

Trial crops deficient in nitrogen, potassium, phosphorus or all three have been shown to have significantly more virus-affected plants than a crop with adequate nutrition.

Inter-row planting into standing wheat stubble significantly reduced virus incidence in small trial plots of PBA HatTrick$^{\Phi}$ compared with the same amount of stubble slashed low to the ground. The mechanism for this difference is unclear, but these results are in agreement with many field observations in large crops during virus outbreaks.

Although differences in virus resistance have been observed for different varieties, further screening is needed to strengthen confidence in these results under high disease pressure in different growing regions, and to identify for which virus species resistance is effective. Under low virus pressure in field trials, some of the better performing varieties included FLIPPER$^{\Phi}$ and PBA HatTrick$^{\Phi}$. although both these varieties have been observed with high rates of infection under high disease pressure. The variety Gully is very susceptible to Ascochyta blight, but has moderate virus resistance so may be useful for breeding resistance into future varieties.

While a link could not be confirmed in the 2013 season between BWYV infections in canola and subsequent spread into nearby chickpea crops, the sometimes high incidence of BWYV in canola indicates it may be prudent to avoid planting chickpea and other pulse crops next to canola. [75,76]

Best agronomic management can help to reduce damage by viruses and includes:

- Retaining standing stubble, which can deter migrant aphids from landing. Where possible, use precision agriculture to plant between stubble rows. This favours a uniform canopy, which makes the crop less attractive to aphids.
- Planting on time and at the optimal seeding rate. These practices result in early canopy closure, which reduces aphid attraction (Figure 10).
- Ensuring adequate plant nutrition.
- Controlling in-crop, fence line and fallow weeds. This removes in-crop and nearby sources of vectors and virus.
- Avoiding planting adjacent to lucerne stands. Lucerne is a perennial host on which legume aphids and viruses, especially AMV and BLRV, survive and increase.
- Seed treatment with insecticides, e.g. imidacloprid, is not effective for non-persistently transmitted viruses but may be effective for luteoviruses. Unfortunately, local data supporting seed treatment are lacking.
- Given the high incidence of BWYV sometimes found in canola, consider growing chickpeas (and other pulse crops) away from canola. [77]

74 K Moore K, A Verrell, M Aftab (2014) Reducing risk of virus disease in chickpeas through management of plant density, row spacing and stubble. GRDC Update Papers 04 March 2014, https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Reducing-risk-of-virus-disease-in-chickpeas

75 Hawthorne 2008. Verrell 2013, 2014. Moore *et al*. 2014.

76 M Sharman, K Moore, J van Leur, M Aftab, A Verrell (2014) Impact and management of viral diseases in chickpeas. GRDC Update Papers 05 March 2014, http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Viral-diseases-in-chickpeas-impact-and-management

77 K Moore, M Ryley, M Sharman, J van Leur, L Jenkins, R Brill (2013) Developing a plan for chickpeas 2013. GRDC Update Papers 26 Feb 2013, http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/02/Developing-a-plan-for-chickpeas-2013

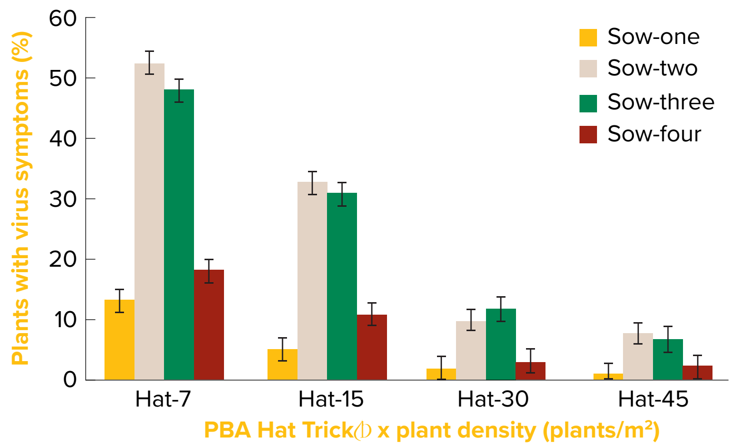


Figure 10: *Proportion of plants with virus symptoms for sowing date by plant density for PBA HatTrick⫯.* [78]

Row spacing and incidence of plants with virus symptoms

Row spacing had a significant effect on incidence of plants with virus symptoms in a 2013 trial. On 11 October 2013, there were more than twice as many symptomatic plants/m^2 in plots with 40 cm rows compared to those with 80 cm rows (Figure 11). Both row configurations were sown at 30 plants/m^2 so plant density per unit area cannot account for the difference. Rather, plant density within each row appears to be responsible (12 plants/m row @ 40 cm and 24 pl/m row @ 80 cm).

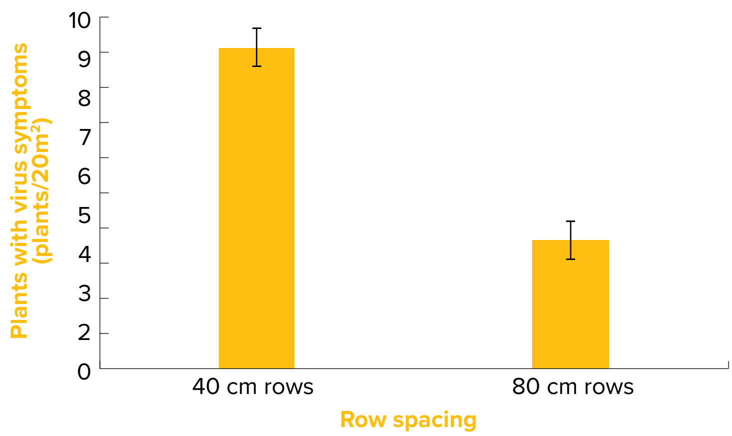


Figure 11: *Effect of row spacing on incidence of chickpea plants with virus symptoms.*

Source: GRDC

Stubble management and incidence of plants with virus symptoms

Planting into standing cereal stubble is known to help reduce risk of virus in lupin crops. Retaining standing winter or summer cereal is believed to be useful in reducing risk of virus in chickpea crops, though research providing such evidence is limited.

78 A Verrell (2013) Virus in chickpea in northern NSW 2012. GRDC Update Papers 26 Feb 2013, http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/02/Virus-in-chickpea-in-northern-NSW-2012

Two trials were conducted in 2013 to compare standing versus flat (slashed) wheat stubble on incidence of plants with virus symptoms. One trial was sown at 80 cm row spacing; the other at 40 cm spacing; both were sown with PBA HatTrick⁽ᵇ⁾ chickpea at 30 plants/m^2. The 80 cm trial was assessed on 11 October and the 40 cm trial was assessed on 9 October and again on 16 October. In both trials, incidence of plants with virus symptoms was lower where the chickpeas had been sown into standing stubble (Figure 12). Individual plots in these trials were small, 2 m × 10 m for the 80 cm trial and 4 m × 10 m in the 40 cm trial. [79]

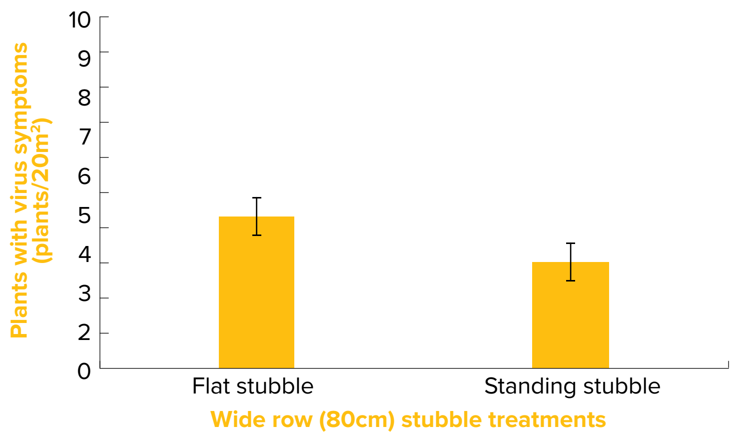


Figure 12: *Effect of stubble management (flat v. standing) on incidence of chickpea plants with virus symptoms.*

Source: GRDC

Non-persistently transmitted viruses (e.g. CMV, BYMV, AMV, and PSbMV)

The initial and main source of infection is contaminated seed, with further transmission in-crop by aphids.

Management steps include:

- Sourcing healthy seed that has been tested free of CMV, BYMV, AMV and PSbMV virus. Tested seed should have less than 0.1% virus infection and field peas should have less than 0.5% for PSbMV.
- Farmer-retained seed should only come from crops with no visible virus symptoms and seed testing should be a priority.
- Some cultivars have virus resistance such as CMV in many new lupin varieties and in JENABILLUP (available in 2011). Yarrum field pea has resistance to BLRV and PSbMV. Increased emphasis on virus resistance is a priority of Pulse Breeding Australia.
- Controlling aphids in-crop is not an effective means of controlling non-persistently transmitted types of viruses.
- Sowing direct into retained cereal stubble and preferably standing as some aphid species are attracted to bare earth. This has been effective in minimising CMV spread in lupins.

Persistently transmitted viruses (e.g. BLRV, BWYV, SCSV)

These viruses are not seed-borne, and the virus is transmitted from live infected plants to healthy plants primarily by aphids or other insect vectors.

79 K Moore K, A Verrell, M Aftab (2014) Reducing risk of virus disease in chickpeas through management of plant density, row spacing and stubble. GRDC Update Papers 04 March 2014, https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Reducing-risk-of-virus-disease-in-chickpeas

An integrated management strategy involves the use of both cultural and chemical measures that aim to eliminate any virus sources, minimise aphids and deter aphids from entering the crop. Often by the time aphids are detected, the virus spread has already occurred.

Management steps include:

- Minimising the 'green bridge' for virus and aphid survival over summer. Control volunteer pulses, legumes and weeds well before sowing and early crop weeds that may carry viruses and aphids.
- Minimising bare earth through sowing into previous cereal stubbles and early sowing with adequate plant population (germ and vigour test seed). Use narrow rows in the absence of stubble to minimise exposed bare soil to deter aphids entering the crop.
- Avoiding crop stress through good paddock selection (soil type, no hard pan, low weed burden) adequate nutrition, no herbicide stresses and good inoculation.
- Avoiding sowing pulses close to each other and broadleaf crops such as canola, and being aware of proximity to perennials (e.g. lucerne).
- Monitoring crops and neighbouring areas using a sweep net or beat sheet. Yellow sticky traps on crop perimeters can also be a handy check for aphid presence. Identify the species present and be prepared to use a 'soft' insecticide such as pirimicarb if there is a chance of localised flights.
- Use of 'soft' insecticides soon after emergence has been shown to help control persistently transmitted viruses only. Use of an SP is controversial as while it prevents early colonisation due to 'anti feed' properties, it can also agitate aphids not controlled and increase virus spread. It should not be used when GPA is present as this major vector for BWYV has resistant populations. Impact on natural beneficials could also lead to higher aphid build-up. [80]

MORE INFORMATION

[Impact and management of viral diseases in chickpeas](#)

[Virus control in chickpea – special considerations](#)

[Managing viral diseases in chickpeas through agronomic practices](#)

80 Pulse Australia (2015) Managing viruses in pulses. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/publications/manage-viruses

Plant growth regulators and canopy management

There are no plant growth regulators for chickpea, but canopy management is a part of chickpea management for pod set and also for disease control.

Factors to consider include time of sowing, row spacing, sowing rate all influence the chickpea canopy. See Sections 3. Planting and 4. Physiology and Plant Growth.

FAQ

Crop desiccation/spray out

Key messages

- Chickpea often matures unevenly and require herbicides to ripen more evenly.
- Desiccation assists production by: taking out late weeds such as thistles which can stain the seed, allowing for earlier harvesting which lessens the weather risk at harvest and browning out green stems which can gum up knives in headers.
- The correct timing for desiccation is when 80–85% of the seeds in the pod have turned yellow and are firm and the remaining 15–20% have yellow 'beaks' on the seed or are starting to turn colour.
- A high water rate is advised to get coverage if using a contact herbicide.
- After desiccation, plants become more brittle, so it is advised not to delay harvesting.

11.1.1 Benefits of desiccation

Desiccation is the strategic termination of crop growth using herbicides. Desiccation is an established technique to improve the rotational fit, benefits and profitability of pulse crops. Desiccation provides important benefits such as reducing weed seed-set, allowing faster harvest and improving grain quality, all leading to improved profitability in pulses.

Desiccation prepares the pulse crop for harvesting by removing moisture from plants and late maturing areas of the paddock. Desiccation is an aid to a timely harvest, particularly where uneven ripening occurs across a paddock, and is now a common practice in lentil and chickpea. Desiccation enables a timely harvest to avoid weather damage.

Crop topping is a form of desiccation, but timing, products and rates differ from desiccation. Desiccation is based on the crop stage close to maturity. The timing of crop topping is based on the stage of development of weed seeds. Different chemicals and rates are used. See Sections 11.3.1 and 11.4.

Application timing is based on the crop when the grain is 75–90% mature, to avoid reducing the quality of the harvested grain. Windrowing can be considered similar to desiccation in timing and benefits to harvest. Windrowing may be considered as an alternative to desiccation. The timing of windrowing is similar to desiccation. [1]

Desiccating a crop overcomes problems with green weeds at harvest and improves harvest efficiency by eliminating many of the problems associated with green stems and gum build-up, such as uneven feeding and drum chokes. Minimising these problems enables drum speeds to be reduced in many cases, with a reduction in cracked or damaged grain. It allows harvesting of a crop that will not naturally shut down due to high soil moisture, and stops chickpeas reshooting and reflowering after pre-harvest rain, and makes crops with uneven maturity more uniform, allowing earlier harvesting. [2]

While desiccation is often not necessary under very hot conditions where the crop is under terminal moisture stress, it can be a very useful harvest management tool in situations where:

- There has been rain during grainfill and the crop is uneven in maturity. Chickpea are very indeterminate and will continue to flower and set up pods late in the season. Crop maturity tends to be very uneven and slow in situations of reasonable moisture supply.

1 Pulse Australia (2015) Desiccation and crop-topping in pulses. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/publications/desiccation-and-croptopping

2 QLD DAFF (2012) Chickpea—harvesting and storage. https://www.daf.qld.gov.au/plants/field-crops-and-pastures/broadacre-field-crops/chickpeas/harvesting-and-storage

- Pod-set has been very uneven due to agronomic factors such as low plant population, poor *Helicoverpa* management, uneven plant establishment in some deep-sown crops, wheel tracks through crops etc.
- There is a problem with actively growing weeds in the crop.

In these situations, desiccation is a valuable management tool for maximising yield and quality through early harvesting. It also improves harvest efficiency by eliminating many of the problems associated with putting green, sappy plant material through the header, i.e. uneven intake and drum chokes. Minimising these problems enables drum speeds to be reduced, with less likelihood of cracking grain. [3]

11.2 Crop-top, desiccate, harvest or manure?

All pulse growers face the decision between crop-topping, desiccation, harvesting or manuring, and their decision is dictated by weed pressures, weed type and the nitrogen demands of the rotation.

11.2.1 When weeds are not the priority

Option 1

Management: natural maturation and grain harvest.

Goal: to maximise grain yield and profit while at the same time providing rotational benefits.

Method: This is the most traditional and widespread practice for cultivating pulses in NSW and is based on well-developed agronomy and crop management strategies from sowing through to harvest. This option assumes weeds are fully managed by conventional rotation and herbicides.

Option 2

Management: brown manuring.

Goal: to maximise N_2 fixation, N-benefit and to conserve soil moisture.

Method: The amount of N_2 fixed is linked closely to dry matter (DM) production of the legume, therefore 'manure' the weed-free pulse close to its maximum DM. For a typical Morgan PSE 23 (long-season) field pea crop sown at Wagga Wagga, NSW, in late May, this would mean desiccating around the end of October.

11.2.2 When weeds are the priority, particularly if herbicide resistance exists

Option 1

Management: brown manuring.

Goal: total control of weeds including herbicide resistance, and to fix some N and conserve soil moisture.

Method: It is imperative to desiccate the crop at or before the milky dough stage of the targeted weed. This often coincides with the flat pod stage of the pulse and inevitably falls well short of the crop's peak DM. At this stage the crop is growing at its maximum rate (about 80–100 kg DM/ha/day), so the amount of N fixed will be proportionally reduced according to its growth stage at desiccation. This cost is non-negotiable and essential to ensure complete weed control.

3 B O'Mara, S Belfield, G Cumming (2007) Chickpea harvest and seed storage. Pulse Australia, www.pulseaus.com.au/storage/app/media/crops/2007_Chickpea-Harvest-Storage.pdf

Option 2

Management: crop-topping/desiccation followed by grain harvest, although this may not be an option available to chickpea because of later crop maturity compared with the weed seeds.

Goal: to maximise grain yield and profit while at the same time providing rotational benefits of preventing weed seed set.

Method: This is the 'have your cake and eat it' scenario. It is a good option for cleaning up scattered weeds and to eliminate weed seed-set in all weedy situations including herbicide resistance. It uses the conventional approach of grain harvest, plus crop-topping/desiccation at the critical growth stage of the weed.

Timing is critical—it depends on the pulse variety reaching physiological maturity at or before the time of crop-topping/desiccation. Most pulse varieties (chickpeas, albus lupin and possibly kaspa field peas) are unsuitable as they are too late and lose too much grain yield. [4] GenesisTM079 is the earliest of the chickpeas, and only infrequently does it mature sufficiently to crop top.

11.3 Timing of desiccation

Chickpea are an indeterminate plant with flowering commencing in the lower canopy, and gradually progressing up the branches (towards the top of the plant) over a 20–30-day period. The problem growers and agronomists are confronted with in the paddock, is how to maximise yield and quality through the optimal timing of the desiccant spray. This can be difficult when you have various stages of seed maturity present on individual plants as well as variability across the paddock.

The optimal stage to desiccate the crop is when the majority (90–95%) of seeds have reached physiological maturity (seeds are below 35% moisture content). The best guide at the present time is to base this on a visual inspection of seeds by cracking open pods on each main fruiting branch. Maximum harvest yield is normally reached when 75% of seeds on each main fruiting branch have turned totally yellow and in various stages of drying down (turning yellow to brown).

Desiccation should occur when:

- Pods in the top 25% of the canopy are mainly in the final stages of grainfill, i.e. where the yellow colouring is moving from the 'beak' down through the seed (Photo 1).
- The bottom 75% of pods have all reached, or dried down below, this stage of maturity. (Seeds have turned totally yellow, and the pod has been bleached to a very light green-yellow colour) (Photo 1).

4 E Armstrong (2015) Weigh up the risks, benefits of pulse harvest. GRDC Ground Cover Issue 115, Profitable pulses and pastures 02 March 2015, https://grdc.com.au/Media-Centre/Ground-Cover-Supplements/Ground-Cover-Issue-115-Profitable-pulses-and-pastures/Weigh-up-the-risks-benefits-of-pulse-harvest

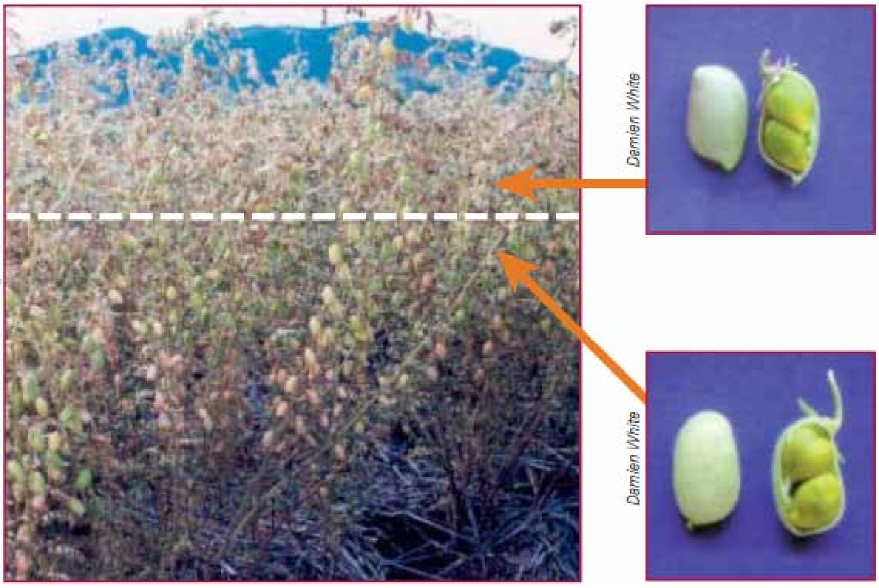


Photo 1: *Chickpea seeds mature progressively from the bottom to the top of the plant.*

Source: Pulse Australia

Monitoring for desiccation timing

Careful monitoring is needed to determine the correct timing for desiccation in both chickpea species. Yield reductions of 10–20% can occur if applied too early. Quality can also be adversely affected. The optimal stage to desiccate chickpea is when the vast majority of seeds have reached physiological maturity i.e. 90–95% of the crop. Inspect the seeds within the upper 20% of pods on each main fruiting branch (Photo 2).

Photo 2: *Correct desiccation timing based on inspection of uppermost pods of each fruiting branch.*

Photo: G Cumming, Pulse Australia

Seeds are considered to be physiologically mature when the green seed colour begins to lighten. The Western Australian recommendation of physiological maturity is 'when the pod wall begins to yellow' (Photo 3, right).

Photo 3: *LEFT: Pods in the top 25% of the canopy should mainly be in the final stages of grainfill, where the yellow colouring is moving from the 'beak' down through the seed. RIGHT: The bottom 75% of pods should have reach maturity. Seeds have turned yellow and the pod has been bleached to a very light green-yellow.*

Photos: G Cumming, Pulse Australia

To avoid the need to inspect seeds, desiccate when 80–85% of pods within the crop have turned yellow-brown (Photo 4). This is usually too late for the control of ryegrass survivors. [5]

Photo 4: *Full maturity, known as 'rattle pod', where the seed has detached from the pod wall and will rattle when shaken.*

Photo: G Cumming, Pulse Australia

Seed and pod development

Chickpea plants are indeterminate and the period of flowering can extend from 20–50 days depending on levels of flower abortion and the impact of moisture stress on the plant. Causes of flower abortion and poor pod-set have been discussed previously and they include:

- low mean daily temperature (below 15°C)
- frost
- Botrytis grey mould
- extended periods of overcast weather.

Flowering commences on the main stem and basal branches, and proceeds upward at intervals of ~2 days between successive nodes on each fruiting branch.

5 Pulse Australia (2015) Desiccation and crop-topping in pulses. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/publications/desiccation-and-croptopping

Under favourable conditions, the time taken from flowering to the visual appearance of the pod (pod-set) is ~6 days. After pod-set, the pod wall grows rapidly for the next 10–15 days to assume full pod size. The seeds start to develop at about the same time as the growth of the pod wall ceases.

Seed growth occurs over the next 20 days. Pod and seed maturation is also very staggered along each fruiting branch, although it is generally more compressed and of shorter duration than flowering owing to the effects of higher temperatures and varying degrees of moisture stress on the plant. The problem faced by agronomists in a commercial paddock situation is how to optimise the timing of the desiccant spray when there are various stages of seed maturity present on individual plants, as well as variation across the paddock. This can be compounded by variation in soil type or paddock micro-relief adding to the problem of uneven crop maturity. Some agronomists use a rule of thumb that when 90% of the field is 90% mature they will advise growers to spray it out. Alternatively, when larger areas are involved, they may split soil types and test them separately for desiccation timing. Often, inspection of commercial crops nearing desiccation reveals that while the lower 30% of pods have dried to below 15% seed moisture (seeds detached from pod and rattle when shaken), the upper 30% of pods on each fruiting branch are still at 30–40% moisture content and in varying stages approaching physiological maturity. [6]

Effect of desiccants on immature seeds

Desiccants should not be applied too early as they can affect green seeds. The result can be a reduction in grain size and yield, an increase in immature seeds, an increase in greenish discolouration of the seed coat and a reduction in seed viability (Table 1). Glyphosate does impact on the normal seedling count in germination tests.. Do not use it in crops destined for sowing seed.

Table 1: *Effects of desiccation timing on seed viability.*

Trial and treatment	Crop stage	% normal seed	% abnormal seed	% dead seed
None	Mature pods	87	9	4
Roundup®	Mature pods	84	14	2
Ally® & Roundup®	Mature pods	85	13	2
Ally® & Roundup®	Mature pods	76	20	4
Ally® & Roundup®	70% green pods	15	63	22
Ally® & Roundup®	All green pods	22	60	18

Source: Qld DPI (1999)

11.3.1 Products for the desiccation of chickpea

1. Reglone® is registered at 2–3 L/ha
2. Reglone® provides quick leaf drydown but the chickpea plant and weeds can quickly regrow if moisture is available
3. Roundup PowerMAX® is the only glyphosate registered for chickpea desiccation
4. a) For chickpea desiccation: Roundup PowerMAX® at 0.68–1.8 L/ha
5. b) For additional weed and chickpea desiccation: Roundup MAX® at 0.5–1.1 L/ha plus Ally® at 5 g/ha
6. Roundup PowerMAX® and Roundup PowerMAX®/Ally® will kill the plants reducing the likelihood of regrowth [7]

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6 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

7 B O'Mara, S Belfield, G Cumming (2007) Chickpea harvest and seed storage. Pulse Australia, www.pulseaus.com.au/storage/app/media/crops/2007_Chickpea-Harvest-Storage.pdf

Table 2: *Chemicals registered for desiccation in chickpeas.*

Herbicide	Example trade names	Operation	Rate	Withholding period
Diquat 200g/L	Reglone®	Desiccation	2 to 3 L/ha	Grazing/stockfeed (GSF): 1 day Harvest: 0 days (lupin, dry pea) 2 days (chickpea, lentil, faba bean)
Paraquat 250g/L	Gramoxone®	Croptopping	400 to 800 mL/ha	GSF: 1 day (7 days for horses) Stock must be removed from treated areas 3 days before slaughter Harvest: 7 days
Glyphosate 480g/L	Ripper 480®	Desiccation	765 mL to 2.025 L/ha	GSF: 7 days Harvest: 7 days
Glyphosate 540g/L	Roundup PowerMAX®	Desiccation	680 mL/ha to 1.8 L/ha	GSF: 7 days Harvest: 7 days
Metsulfuron + Glyphosate 540 g/L	Ally® + Roundup PowerMAX®	Desiccation + knockdown weed control	5 g + 500 mL to 1.1 L/ha	GSF: 7 days Harvest: 7 days
Saflufenacil	Sharpen®	Desiccation	34 g/ha plus recommended label rate of glyphosate or paraquat herbicide plus 1 % Hasten or high quality MSO	GSF: 7 days Harvest: 7 days

GSF - Withholding period for grazing or cutting for stock food

Note: Observe the Harvest Withholding Period and GSF for each crop.

Source: Pulse Australia

Paraquat is registered for crop-topping; however, may not be effective on grass seed-set as chickpeas mature quite late relative to grasses.

The major differences between timing of desiccation and crop-topping are:

- application timing is different and initiated by different criteria
- herbicides for crop-topping and desiccation are not always the same
- herbicide rates for desiccation are higher than that required for crop-topping
- crop-topping will advance the harvest timing in some pulse crops
- neither desiccation nor crop-topping can be used effectively in all pulses
- both will cause reduced grain quality and yield if applied at the wrong maturity stage of the crop. [8]

NOTE: Desiccation can affect seed viability if applied incorrectly. To avoid damaging seed viability, it is advisable not to desiccate or crop-top a pulse seed crop.

11.4 Crop-topping

Crop-topping is timed to prevent weed seed-set, not by the crop growth stage. Hence, crop-topping is generally not possible in chickpea, as they are too late in maturing. Crop-topping chickpeas can result in discoloured cotyledons (kernel) and seed coats, leading to rejection at delivery and/or severe downgrading. Even in other

8 Pulse Australia (2015) Desiccation and crop-topping in pulses. Pulse Australia, Australian Pulse Bulletin, http://www.pulseaus.com.au/growing-pulses/publications/desiccation-and-croptopping

pulses, growers need to be aware of grain quality defects if crop-topping is done earlier than the crop desiccation or windrowing stage.

Genesis™ 079 is the earliest maturing chickpea variety, but in most cases, it will not mature early enough to enable efficient crop-topping without grain quality impacts. Evidence of the lack of suitability of crop-topping in chickpea is provided in Table 3, from a South Australian Research and Development Institute crop-topping trial at Melton, South Australia, in 2009. Visual grain quality data are not presented, but in this trial:

- Many responses to crop-topping treatments may have been masked by rapid senescence from a rapid, early seasonal finish (e.g. Almaz⦶ and Genesis™ 114).
- When crop-topped at the recommended stage, yields were 69–86% of the untreated control (31–14% yield loss). When crop-topped 2 weeks after the optimum stage for ryegrass, yields were 92–114% of the untreated control. When crop-topping was 3 weeks ahead of the recommended ryegrass stage, yields were 17–48% of the untreated control (83–52% yield loss).

Table 3: *Impact of crop-topping timing on chickpea varieties of differing maturity compared with an untreated control at Melton, South Australia, 2009 Pink shading denotes significant difference from the control treatment.*

	Control yield (t/ha)	Yield (% of control) for each timing			Control grain weight (g/100 seeds)	Grain weight (% of control) for each timing		
		Minus 3 weeks (9 Oct.)	Recommended ryegrass control stage (30 Oct.)	Plus 2 weeks (12 Nov.)		Minus 3 weeks (9 Oct.)	Recommended ryegrass control stage (30 Oct.)	Plus 2 weeks (12 Nov.)
Almaz⦶	1.18	19	83	92	27.4	91	92	91
PBA Slasher⦶	1.96	30	70	99	15.5	87	84	100
PBA HatTrick⦶	1.37	36	69	85	18.1	77	81	93
Genesis™ 079	2.09	25	80	107	18.0	95	104	104
Genesis™ 090	1.43	25	84	97	22.1	79	93	93
Genesis™ 114	0.90	17	86	114	22.1	96	102	104
Genesis™ 509	1.96	32	71	96	13.6	129	101	94
Howzat⦶	1.70	21	72	94	16.6	87	87	117
Sonali	2.13	40	77	104	14.5	96	80	101
Mean (t/ha)	1.90	0.6	1.5	1.90	18.6	16.3	15.9	18.2
Mean (g/100)					18.6	16.3	15.9	18.2

Source: M Lines and L McMurray (SARDI), Southern Pulse Agronomy Research trials

Harvest

Key messages

- Greatly improved crop management and harvest timing has meant that chickpeas can be harvested earlier, with associated yield and marketing benefits. The tradition of delaying the harvest of chickpeas until after wheat can result in considerable chickpea yield and quality losses.
- Early, or timely, harvest of the chickpea crop has the potential to increase returns by up to 50%.
- If harvesting grain for seed, germination rates are improved if grain is harvested at 12–14% (Photo 1), and then stored in aerated silos or immediately graded and bagged.
- During harvest, chickpeas can produce a dust which is quite flammable, so make sure headers are blown down frequently to avoid fire.

Photo 1: *Chickpea harvest under way.*

ABC Rural

12.1 Windrowing and swathing

Windrowing of chickpeas is possible, but it is not widely used because there is little or no stubble for the windrow to sit on as there is, for example, with canola. Losses at harvest may be greater, and more dirt may enter the grain sample. Light windrows can be blown away in strong winds. Despite this, provided the windrows are large enough and compacted, windrowing is possible. It may also be possible to place two swathes into the one windrow and compact it with a cotton reel roller when windrowing. This technique shortens harvesting time.

In chickpeas, windrowing or desiccation can occur when <20% of pods are green and 90% of seed is changing from a green colour. The main advantages of windrowing are earlier harvest, reduced seed damage and less shattering or pod loss, particularly if harvest is delayed. Pod loss and shatter are reduced because windrowers allow unhindered passage onto the canvas due to the absence of platform augers. Lower harvesting heights may also be possible.

Windrowing also helps to dry out green broadleaf weeds, such as radish, which can cause major problems at harvest.

Windrowing also reduces damage to headers. Sticks and stones in rougher country can damage knife fingers and sections, retractable fingers and other components of headers, but pick-up fronts leave most of these on the ground. The cutting height for windrowing should be just below the bottom pods, with the reel following the top of the crop. The reel speed should be quite slow. The delivery opening in the windrower should be large enough to prevent blockages; otherwise, there will be lumps in the windrow. Windrows should be dense and tightly knit for best results.

Curing should take about 10 hot days. However, heavy infestations of radish and other weeds could delay drying. Pick-up fronts are the most common type used for harvesting windrows. However, crop lifters used close together on open fronts have been used with some success. [1]

12.2 Harvest timing

Chickpea harvest can often clash with wheat harvest, and traditionally wheat has been given priority due to potential quality premiums. However, this thinking needs to be balanced against the relatively higher value and potential yield and quality losses that can result from a late chickpea harvest. Agronomists report that many growers consider losses in chickpeas will generally be less than in cereals. However, yield losses increase significantly the longer harvest is delayed.

Chickpeas should be harvested as soon as they mature (Photo 2), as pods will fall if harvest is delayed. [2] Crop desiccation enables even earlier harvest. [3]

Photo 2: *Mature chickpea plant.*

Source: The Land

Harvesting early also minimises infection of seed. Diseases can be transmitted in stubble and soil, and on machinery and boots. Soil and stubble can be moved by machinery, during windy or wet weather, and in floodwater. To reduce the transmission of diseases, clean headers, sowing equipment and spray rigs to remove grain, soil and stubble before moving from property to property. [4]

Harvest timing will depend on the moisture content that is acceptable for delivery or storage. This will depend on who is buying the grain, or whether aeration is available in the storage. Harvesters should be set up to operate efficiently at 14–15% grain MC. This effectively doubles the harvest period available on any one day compared to harvesting at 12%. Research has shown that average harvest losses increased as harvest was delayed (and seed moisture decreased). [5]

1 Pulse Australia (2013) Northern Chickpea—Best Management Practices Training Course Manual 2013. Pulse Australia.

2 GRDC (2008) Harvesting. In Grain Legume Handbook. GRDC, https://grdc.com.au/resources-and-publications/all-publications/publications/2008/03/2008-grains-legume-handbook

3 GRDC (2013) Chickpea disease management (southern and northern regions). Fact sheet. GRDC, http://www.grdc.com.au/Resources/Factsheets/2013/05/Chickpea-disease-management

4 GRDC (2013) Chickpea disease management (southern and northern regions). Fact sheet. GRDC, http://www.grdc.com.au/Resources/Factsheets/2013/05/Chickpea-disease-management

5 B O'Mara, S Belfield, G Cumming (2007) Chickpea harvest and seed storage. Pulse Australia, http://www.pulseaus.com.au/storage/app/media/crops/2007_Chickpea-Harvest-Storage.pdf

The maximum moisture for chickpeas is 14% for grower receivals. Harvesting grain at 13–15% moisture content will help to minimise cracking. Above 14% moisture, the crop should be either aerated or dried. Aeration is usually very effective in reducing chickpea moisture content by several percentage points. [6]

Harvesting at moisture levels below the receival standard of 14% can be costly. Moisture content decreases with late harvest (Table 1). [7] Delaying harvest from 14% MC to 8% MC for a 500 tonne crop equates to a 32 tonne weight reduction, and a loss of $17,500 (at $550/t). This is in addition to any harvest losses that occur due to low moisture at harvesting.Pulse Australia has calculated the economic losses caused by loss of moisture below the Grain Trade Australia (GTA) receival standard of 14% moisture content (MC) maximum.

- 500 t of chickpea at 14% grain moisture, at $450/t, is worth $225,000.
- The same grain harvested at 8% moisture delivers 470 t, at $450/t, and is worth $210,600.
- This is a loss to the grower of $14,400. [8]

Table 1: *Yield and moisture loss with delayed harvest.*

Harvest timing	Average moisture	Harvest loss
On time	12.7	10%
Late	10.3	23%

Source: Pulse Australia

Note: Crops intended for seed are best harvested at 14–16% MC and dried or aerated back to 12% to maximise both germination and vigour when held in storage. [9]

Yield losses of up to 30% have been recorded in the field, due to delayed harvest (Figure 1). Grain losses due to a 2–4 week delay in harvest were estimated at A$93–238/ha, depending on seasonal conditions. In this instance, most of these losses were due to pod loss at the header front, or unthreshed pods discarded out of the back of the machine.

In most years, chickpea yields can average ~70% of wheat yields when sown in an identical situation. The use of specialised headers and separate storage facilities for chickpeas may alleviate the competition with wheat for time, labour and equipment usage.

6 DAF Queensland (2012) Chickpea—harvesting and storage. Department of Agriculture and Fisheries Queensland, https://www.daf.qld.gov.au/plants/field-crops-and-pastures/broadacre-field-crops/chickpeas/harvesting-and-storage

7 B O'Mara, S Belfield, G Cumming (2007) Chickpea harvest and seed storage. Pulse Australia, http://www.pulseaus.com.au/storage/app/media/crops/2007_Chickpea-Harvest-Storage.pdf

8 Pulse Australia (2013) Northern Chickpea—Best Management Practices Training Course Manual 2013. Pulse Australia.

9 B O'Mara, S Belfield, G Cumming (2007) Chickpea harvest and seed storage. Pulse Australia, http://www.pulseaus.com.au/storage/app/media/crops/2007_Chickpea-Harvest-Storage.pdf

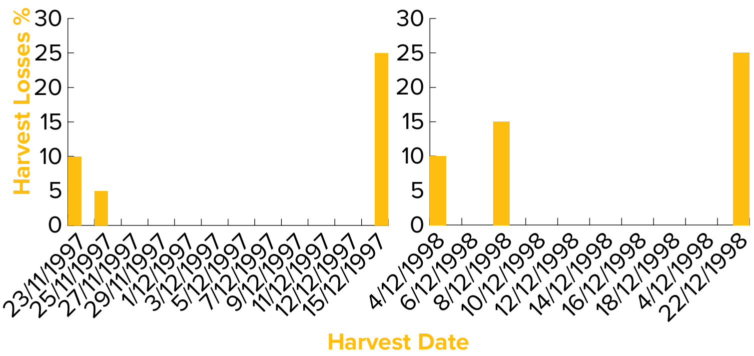

Figure 1: *Harvest yield losses in 1997 (left) and 1998 (right).*

Source: Pulse Australia

Although not normally prone to pod splitting and shelling-out in all but extreme wet-weather conditions, chickpeas are very prone to pod-drop as the plant dries down. Prolonged weathering in the field weakens the hinge attaching the pod to the stalk, thus increasing pod-drop both before and at harvest, and causing drops in yield.

Lodging is increasingly likely the longer chickpeas are left in the field. The risk is higher if the crop is high yielding and has been planted on wide rows of 70–100 cm.

Increased storage is helping farmers to manage losses, and in some instances, reduce freight and handling costs where direct transport of grain to the end-user is possible.

12.2.1 Major losses from late harvest

Major losses incurred by harvesting chickpeas late include loss of yield, loss of quality, greater likelihood of disease and insect damage to pods and seeds, and loss of markets.

Loss of yield

- Losses due to pod drop can be severe as weathering weakens the hinge attaching the pod to the stem.
- Weathered pods become more difficult to thresh, resulting in grain loss from unthreshed pods passing out the back of the header, increased numbers of cracked grains, and a slower harvest.
- Increased lodging, especially in higher-yielding crops that are planted on wide rows.
- Harvesting at 8% MC instead of 14% results in a harvest loss.
- Farmer experience has shown yield losses of up to 30% if harvest is delayed 2–4 weeks.

Loss of quality

- Weathered or very dry grain is more likely to crack when handled, increasing the amount of split grain in the sample. Levels of cracked and damaged grain can be as high as 50% in extreme cases of field weathering and prolonged rainfall.
- The number of unthreshed pods in the sample will increase, as they become harder to thresh with weathering.
- Both split grain and unthreshed pods can result in rejection or the need for grading to meet market requirements.
- The germination rate and vigour of planting seed will be reduced by weathering.

- Chickpea grain discolours and darkens with weathering, reducing its marketability, particularly in the container market. The following conditions play a major role in accelerating seed-coat darkening (Photo 3):
- rainfall
- cool–mild temperatures
- high humidity
- Although there is usually no direct penalty or discount for a moderate degree of seed-coat darkening, it does have a significant impact on the marketability of the product and the reputation of the Australian industry as a supplier of quality product. Quality is becoming increasingly important as Australian traders attempt to establish market share against other chickpea-exporting countries such as Canada, Turkey and Mexico. We will likely see much greater segregation and premiums paid for lighter-coloured, large-seeded desi types as new varieties with these traits are developed and the Australian industry becomes more quality conscious.

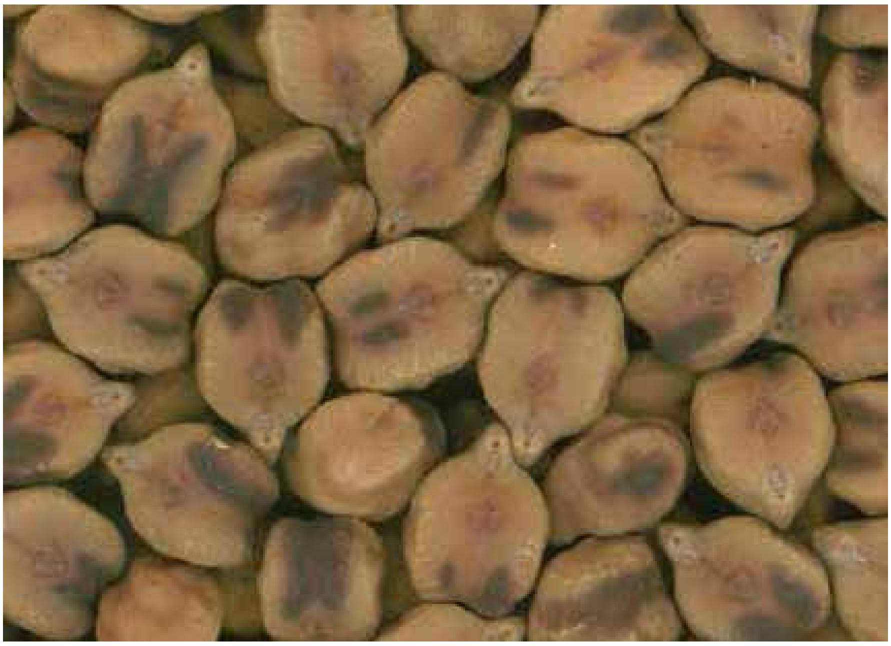

Photo 3: *PBA Pistol*$^{\oplus}$ *chickpea showing grain discolouration due to weathering.*

Photo: Jenny Wood, NSW DPI

Note: Chickpeas that do not meet the export receival standard of 6% maximum 'defective' chickpeas need to be graded. This incurs a grading cost to the grower of $15–25/t. Downgrading into the stockfeed market results in a value of $120–140/t.

Increased disease and insect risk to pods and seed

- Weathering of seed due to delays in harvest can greatly increase levels of mould infections. High levels of mould infection will also cause darkening of the seed coat.
- Humid (>70% relative humidity), wet conditions favour the development of a range of fungi in late-harvested chickpea crops. *Alternaria* spp. usually predominate, species of *Asperguillus*, *Cladosporium* and *Penicillium* may also be present.
- There is increased risk of late infection by the Ascochyta blight fungus on pods. Ascochyta blight can infect senescing pods under wet conditions, leading to infected and discoloured seed (and possible rejection). The current export receival standard for visible Ascochyta blight lesions is a maximum of 1% on the seed cotyledon (kernel).

- Native budworm (*Helicoverpa punctigera*) can cause damage to mature seeds. Larvae can occasionally attack senescing chickpeas, particularly where rainfall has softened the pod. Insect-damaged seeds are classified as defective, and they cannot exceed the tolerance level of 6%.

Lost marketing opportunities

- Chickpea prices can reach peaks during harvest, when demand is higher to meet shipping schedules. Earlier harvesting may allow access to these opportunities.
- Early harvest gives the grower some control over how and when the crop is marketed, whereas growers of late-harvested chickpeas are more likely to end up being 'price-takers' in a falling market.
- Darker, weathered seed may be discriminated against in the market.

Harvest delays in chickpeas cost growers and the pulse industry a lot of money. In any production area, a spread of 4–6 weeks may occur in the harvesting of chickpea crops planted on the same sowing rain. Many of the late-harvested crops have moisture content down to ~8%, a big drop from the maximum moisture content for receival of 14%, and the preference for 12%. [10]

12.2.2 Planning for early harvest

Early, or timely, harvest of the chickpea crop has the potential to increase returns by up to 50%. Management to ensure even crop maturity and timely harvest consists of a combination of factors, including:

- paddock selection and agronomy
- disease and insect control
- desiccation
- harvest timing and technique
- handling and storage [11]

These influence the management options that contribute to an early harvest. They will all be important at different times and for different reasons. It is important to understand the potential and limitations of each component in how a crop is managed. Optimal results in terms of yield, profit and earliness will be due to these components being applied in the most appropriate and balanced way, and as dictated by seasonal conditions. The components include the following.

1. Planting:

- Sow at the earliest opportunity within the preferred planting window for your area. Moisture-seeking equipment and/or press wheels can significantly enhance seeding opportunities under marginal conditions.
- Select adapted varieties that meet your target for early harvesting.
- Using precision planters will often achieve more uniform plant establishment and crop development and, consequently, more even crop maturity. Precision planters are not widely used in the south but there is growing interest in them.

2. In-crop management:

- Control Botrytis grey mould if present during flowering.
- Control native budworm during flowering to maximise early podset.
- Avoid using herbicides such as flumetsulam (e.g. Broadstrike®) that delay crop maturity..

3. Harvest management:

- Consider using Roundup UltraMAX® and Ally® (or equivalent registered products) to terminate the crop at 80–90% yellow–brown pod stage.
- Set up the header to operate efficiently at 14–15% grain moisture content.

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10 Pulse Australia (2013) Northern Chickpea—Best Management Practices Training Course Manual 2013. Pulse Australia.

11 B O'Mara, S Belfield, G Cumming (2007) Chickpea harvest and seed storage. Pulse Australia, http://www.pulseaus.com.au/storage/app/media/crops/2007_Chickpea-Harvest-Storage.pdf

- A major advantage of high-moisture harvesting is that harvest can commence earlier in the season and earlier each day: harvesting at 14% MC, compared with 12%, can effectively double the harvest period available on any one day.
- Blend, aerate and/or dry the sample to the required receival standard of 14% MC. [12]

12.3 Header modifications and settings

Early harvesting means that plants can be easier to gather because they stand more erect, allowing the harvester front to operate at a greater height, reducing the amount of soil, rock and sticks that enter the harvester. Early harvesting also means there are fewer summer weeds to clog the harvester. Grain loss can be reduced by harvesting in high humidity, at night if necessary, to minimise pod shattering. Avoid reaping in extreme heat. [13]

Chickpeas can be harvested with minor adjustments and modifications to equipment. Open-front or pick-up fronts are best suited to the job. Pulses are easily threshed, so concave clearances should be opened and the drum speed reduced. The crop varies in height from 15–80 cm, with pods held up in the canopy, so direct heading without crop lifters is possible with open-front and closed-front machines. Some fingers may have to be removed when using closed-front machines. Chickpeas thresh easily but are prone to cracking, particularly kabuli types, so adjust thresher speed to 400–600 rpm and the concave to 10–30 mm to suit (Table 2). Because chickpeas are destined for human consumption, a good sample off the header is usually required. [14]

Table 2: *Harvester settings for pulses.*

Reel speed	Spiral clearance	Thresher speed	Concave clearance	Fan speed	Top sieve	Bottom sieve	Rotor speed
Medium	High	400–600 rpm	10-30 mm	High	32 mm	16 mm	700–900 rpm

Source: Grain legume handbook.

A straw chopper may be of value to chop up the stubble and spread it uniformly.

Crop lifters are not usually required unless the crop is badly lodged. Set the finger-tine reel to force the chickpea material down onto the front. Moving the broad elevator auger forward can improve the feeding of light chickpea material. Vibration from cutter-bar action, plant-on-plant or reel-on-crop impact, and poor removal of cut material by the auger all cause shattering and grain loss. Finger reels are less aggressive than bat reels and cause fewer pod losses. Double-acting cutter bars reduce cutter-bar vibration losses. Four-finger guards with open second fingers also reduce vibrations (Figure 2). [15]

12 Pulse Australia (2013) Northern Chickpea —Best Management Practices Training Course Manual 2013. Pulse Australia.

13 Pulse Australia (2013) Northern Chickpea —Best Management Practices Training Course Manual 2013. Pulse Australia.

14 GRDC (2008) Harvesting. In Grain Legume Handbook. GRDC, https://grdc.com.au/resources-and-publications/all-publications/publications/2008/03/2008-grains-legume-handbook

15 Pulse Australia (2013) Northern Chickpea —Best Management Practices Training Course Manual 2013. Pulse Australia.

Figure 2: *Finger guard.*

Source: Grain Legume Handbook

12.3.1 Options to improve harvest

- Vibra-mat: a vinyl mat that vibrates with the knife, stops bunching at the knife of open-front headers and helps the table auger to clear-cut materials. This device is very cheap. It is more effective in light crops. It is important to match ground speed to table auger capacity and crop density—too slow and the plants will not have enough momentum to carry to the front; too fast and the cut crop will not be cleared from behind the knife.
- Extension fingers (Photo 4): plastic extension fingers ~30 cm long that fit over existing fingers. Extension fingers can save significant losses at the knife for little financial outlay. Pods that would have fallen in front of the knife are caught on the fingers and pushed into the comb by the incoming crop.
- Extended fronts are now available for some headers. They reduce losses at the knife by increasing the distance between the knife and auger to a maximum of 760 mm. This helps to stop material bunching in front of the auger, where pods can fall over the knife and be lost.
- Platform sweeps are used in conjunction with extended fronts. They consist of fingers that rake material towards the auger to help eliminate bunching. They can also be used on conventional fronts.

Note that costs and benefits must be assessed; a small area of pulses may not justify the cost of some of these modifications. [16]

16 GRDC (2008) Grain Legume Handbook. GRDC.

Photo 4: *Plastic extension fingers fitted to a draper front.*

Photos: G. Cumming, Pulse Australia

Draper fronts

Draper fronts (e.g. MacDon or Honeybee) have become increasingly popular. The centre-feed draper platform provides uniform crop flow into the header, with minimal crop loss and little damage to the seed. The cutter-bar design allows for both vertical and end-table flotation. While their contour following ability is not quite as good as a floating cutter bar, they have performed very well, provided the paddock is relatively level. Operators claim they can be operated at higher travel speeds than a conventional front in chickpeas.

Preferred air-front set-ups

Air fronts help to reduce shattering losses, and minimise the amount of soil and other debris (e.g. stubble, sticks) in the final sample. Where soil contamination is likely to be a problem, fit perforated screens to replace the feeder-house floor and elevator doors, and clean the grain cross-augers. Twin blowers may be necessary on fronts wider than 7.6 m. [17]

- Harvest-Aire or other air fronts are generally considered better than batt reels as they minimise the risk of pods detaching from the plant.
- They also improve feed in over the knife section, and reduce soil and stubble contamination, and allow the operator a clearer view of the cutting platform, as well as any rocks or sticks in the paddock. Adjustment of the angle and height of the air nozzles is critical, and may need adjustment as crop conditions change.
- Fit a Vibra-mat to improve the flow of material over the knife-section and along the platform. They are relatively cheap to buy and to maintain.
- Fit cast, short-crop fingers. If using a closed front the fingers will need to be spaced 19 mm or more apart.
- Fitting double-density Kwik-cut knife guards will help reduce plant vibration and the risk of pods detaching from the plant. This method may be unsuitable if there are a lot of green weeds in crops that are not desiccated, as the weeds will cause blockages.
- Check that the header front is level, and not higher at one end than the other. Set the knife at the correct angle for short crops, and install a simple depth gauge.
- In crops with a short height to the lowest pods, soil contamination is likely to be a problem, so it is advisable to fit perforated screens under the platform auger and/or broad elevator. Fit screens to repeat and clean-grain cross-augers.

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17 DAF Queensland (2012) Chickpea—harvesting and storage. Department of Agriculture and Fisheries Queensland, https://www.daf.qld.gov.au/plants/field-crops-and-pastures/broadacre-field-crops/chickpeas/harvesting-and-storage

- Floating or flexible cutter bars can be useful in short crops.

Conventional headers

- Aim to harvest at 300–500 rpm where possible to minimise cracking. Adjust upwards if jamming occurs in crops that are not desiccated.
- Set concave clearance at 10–30 mm depending on seed size. Check the concave for uneven clearance. Standard concaves tend to bow in the centre when fully loaded, and may need strengthening or replacement (e.g. with a Loewen concave). Removing alternate wires and the blank-off plates from the concave will also help reduce cracking. If possible, cover the rasp bars with plate.
- Beater: reduce speed to 100% of drum speed (for wheat it is usually set at 150%.)
- Set fan speed at 80–100% of maximum. The relatively heavy weight of individual chickpea grains allows the use of high air flow. [18]

Sieves

An alternative to the barley sieve is a mesh sieve made using 18-mm tubing for the frame and 1 cm by 1 cm, 14-gauge wire mesh. This screen increases capacity because the whole area is able to sieve. If there are summer weeds, the rake at the back of the sieves should be blanked-off to stop them entering the returns. Summer weeds may cause walkers and sieves to block completely, causing high grain loss. [19]

Set sieves to suit the grain size of the chickpea being harvested. This is more critical than for wheat:

- Top sieve 20–25 mm—a B & D Airfoil non-adjustable top sieve is reported to work well in chickpeas, and increases overall sieving capacity.
- Bottom sieve 12–16 mm—the bottom sieves can be altered so that the front 400 mm can be adjusted separately to the rear section. This allows the front section to be left open, and more air can be directed onto the top sieve if required.

Header speeds

Relatively slow ground speeds are considered essential when harvesting chickpeas to minimise excessive losses at the front of the header and the amount of dirt entering the machine.

- A maximum speed of 8 km/h is recommended.
- If using a batt reel, it should be set at the same speed as the header. [20]

12.4 Getting a clean sample

The harvesting of chickpeas can be costly if stones, sticks or too much soil are picked up with the chickpeas. Machinery damage can be reduced by a variety of practices.

12.4.1 Perforated screens

Perforated screens fitted on the bottom of the broad elevator, cross-augers, and grain and seconds elevators all reduce the amount of soil in the sample. The perforated screen at the broad elevator is large, and removes soil before it enters the main working mechanism of the harvester.

12.4.2 Harvester speed

Excessive harvester speeds will cause large losses of grain and force more soil into the harvester. Generally, speeds >8 km/h are not recommended, irrespective of the type of harvester front used.

18 B O'Mara, S Belfield, G Cumming (2007) Chickpea harvest and seed storage. Pulse Australia, http://www.pulseaus.com.au/storage/app/media/crops/2007_Chickpea-Harvest-Storage.pdf

19 Pulse Australia (2013) Northern Chickpea—Best Management Practices Training Course Manual 2013. Pulse Australia.

20 B O'Mara, S Belfield, G Cumming (2007) Chickpea harvest and seed storage. Pulse Australia, http://www.pulseaus.com.au/storage/app/media/crops/2007_Chickpea-Harvest-Storage.pdf

12.4.3 Harvesting in high humidity

Harvesting in humid conditions, when pods are less prone to shatter, can reduce grain losses. However, more unthreshed pods may appear in the grain sample. It is unwise to harvest chickpeas at night unless using a pick-up front or some positive height control, which will stop the front from digging into the soil. Some farmers have fitted wheels on the outer end of their fronts, as a depth stop. Others have purchased ultrasonic automatic depth controls to control header height.

12.4.4 Pick-up fronts

Pick-up fronts that are the same as, or similar to, those used for picking up windrows can be used to harvest windrowed chickpeas. Pick-up fronts greatly reduce the amount of soil entering the harvester and make harvesting easier because harvesting height is not as critical as with a front fitted with lifters. They allows harvesting at night. The fingers on the pick-ups are closely spaced and they will gather the entire crop, so crop losses are reduced.

There are different types of pick-ups. Some have fingers attached to rotating belts (draper pick-ups) and others have fingers attached to rotating drums (peg-roller pick-ups). The peg-roller types are similar and cheap, but tend to shatter pods and cause slightly higher grain losses than the draper type. The draper types are more expensive but will reduce losses if harvesting late.

12.4.5 Flexible cutter-bar fronts (flexi-fronts)

The cutter-bars of flexi-fronts are hinged in short sections, allowing the whole front to flex and closely follow the ground contour. They use skid plates and are particularly good for short crops such as lentils and peas, but can also be used on cereals by locking the hinged sections together. [21]

12.4.6 Lodged crops

If the crop has lodged, the best option is usually to harvest directly into, or at right angles to, the direction the crop has fallen. If on wide rows, use crop lifters and harvest up and back in the rows. The crop usually feeds in better over the knife section, and also provides the header operator with a better view of any rocks or sticks in the paddock. [22]

12.5 Fire prevention

Grain growers must take precautions during the harvest season, as operating machinery in extreme fire conditions is dangerous. They should take all possible measures to minimise the risk of fire. Fires are regularly experienced during harvest in stubble as well as standing crops. The main cause is hot machinery combining with combustible material. This is exacerbated on hot, dry, windy days. Seasonal conditions can also contribute to lower moisture content in grain and therefore a greater risk of fires.

During harvest, chickpeas can produce a dust which is quite flammable, so make sure headers are blown down frequently to avoid fire.

12.5.1 Harvester fire reduction checklist

1. Recognise the big four factors that contribute to fires: relative humidity, ambient temperature, wind, and crop type and conditions. Stop harvest when the danger is extreme.

21 GRDC (2008) Grain Legume Handbook. GRDC.

22 Pulse Australia (2013) Northern Chickpea—Best Management Practices Training Course Manual 2013. Pulse Australia.

2. Focus on service, maintenance and machine hygiene at harvest on the days more hazardous for fire. Follow systematic preparation and prevention procedures.
3. Use every means possible to avoid the accumulation of flammable material on the manifold, turbocharger or the exhaust system. Be aware of side and tailwinds that can disrupt the radiator fan airblast that normally keeps the exhaust area clean.
4. Be on the lookout for places where chaffing can occur, such as fuel lines, battery cables, wiring looms, tyres and drive belts.
5. Avoid overloading electrical circuits. Do not replace a blown fuse with a higher amperage fuse. It is your only protection against wiring damage from shorts and overloading.
6. Periodically check bearings around the harvester front and the machine. Use a hand-held digital heat-measuring gun for temperature diagnostics on bearings and brakes.
7. Maintain fire extinguishers on the harvester and consider adding a water-type extinguisher for residue fires. Keep a well-maintained firefighting unit close-by to the harvesting operation ready to respond.
8. Static will not start a fire but may contribute to dust accumulation. Drag chains or cables may help dissipate electrical charge but are not universally successful in all conditions. There are some machine mounted fire-suppression options on the market.
9. If fitted, use the battery isolation switch when the harvester is parked. Use vermin deterrents in the cab and elsewhere, as vermin chew some types of electrical insulation.
10. Observe the Grassland Fire Danger Index (GFDI) protocol on high fire risk days.
11. Maintain two-way or mobile phone contact with base and others, and establish a plan with the harvest team to respond to a fire if one occurs. [23]

Using machinery

To preventing machinery fires, it is imperative that all headers, chaser bins, tractors and augers be regularly cleaned and maintained. All machinery and vehicles must have an effective spark arrester fitted to the exhaust system. To prevent overheating of tractors, motorcycles, off-road vehicles and other mechanical equipment, all machinery needs to be properly serviced and maintained. Firefighting equipment must be available and maintained—it is not just common sense, it is a legal requirement.

Take great care when using this equipment outdoors:

- Be extremely careful when using cutters and welders to repair plant equipment. This includes angle grinders, welders and cutting equipment.
- Ensure machinery components, including brakes and bearings, do not overheat. These components can drop hot metal onto the ground and start a fire.

Use machinery correctly, as incorrect usage can cause it to overheat and ignite.

Be aware that when blades of slashers, mowers and similar equipment hit rocks or metal, they can cause sparks to ignite dry grass.

Avoid using machinery during inappropriate weather conditions of high temperatures, low humidity and high wind.

Do repairs and maintenance in a hazard-free, clean working area, such as on bare ground, on concrete, or in a workshop, rather than in the field.

Keep machinery clean and as free from fine debris as possible, as this can reduce onboard ignitions. [24]

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23 Barr R. (2015). Plant of attack needed for harvester fires. https://grdc.com.au/Media-Centre/Media-News/South/2015/10/Plan-of-attack-needed-for-harvester-fires

24 NSW Rural fire Service. Farm firewise. NSW Government, http://www.rfs.nsw.gov.au/dsp_content.cfm?cat_id=1161

With research showing an average of 12 harvesters burnt to the ground every year in Australia (Photo 5), agricultural engineers encourage care in keeping headers clean to reduce the potential for crop and machinery losses.

Key points

- Most harvester fires start in the engine or engine bay.
- Other fires are caused by failed bearings, brakes and electricals, and rock strikes. [25]

Photo 5: *GRDC figures show that there are 1000 combine harvester fires in Australia each year.*

Source: Weekly Times

12.5.2 Harvesting in low-risk conditions

Growers can use the Grassland Fire Danger Index (GFDI) to assess the wind speed at which harvest must cease (a GFDI of 35), depending on the temperature and relative humidity (Figure 3).

- Step 1: Read the temperature on the left-hand side.
- Step 2: Move across to the relative humidity.
- Step 3: Read the wind speed at the intersection. In the worked example, the temperature is 35°C and the relative humidity is 10 per cent so the wind speed limit is 26kph.

25 GRDC (2012) A few steps to preventing header fires. GRDC Ground Cover Issue 101, http://www.grdc.com.au/Media-Centre/Ground-Cover/Ground-Cover-Issue-101/A-few-steps-to-preventing-header-fires

PODCAST

GRDC podcasts: Harvester Fires

MORE INFORMATION

GRDC Reducing harvester fire risk: The Back Pocket Guide

An investigation into harvester fires

Plan of attack needed for harvester fires

Standards and charts to check visual quality of desi chickpeas are available from Pulse Australia

Australian Pulse Standards 2016–2017

Desi chickpea sample chart of visual standards

TEMP °C	5	10	15	20	25	30	40	50	60	65	RH%*
15	31	35	38	40	43	45	49	53	56	58	AVERAGE WIND SPEED (KPH)
20	29	33	36	38	40	43	46	50	53	55	
25	27	30	33	36	38	40	44	47	50	52	
30	25	28	31	33	35	37	41	44	47	49	
35	23	26	28	31	33	35	38	41	44	46	
40	21	24	26	28	30	32	35	39	41	43	
45	19	22	24	26	28	30	33	36	39	40	
TEMP °C	5	10	15	20	25	30	40	50	60	65	RH%*

*RH% (Relative Humidity rounded down)
†Wind speed averaged over 10 minutes

Figure 3: *Grassland Fire Danger Index guide.*

Source: CFS South Australia

12.6 Receival standards

National receival standards for chickpea are set by the pulse industry and maintained by Pulse Australia. Receival and export standards reflect the market requirements for a quality food product. Desi chickpeas should be sound, dry, fresh and light to medium brown in colour, although a greenish tinge is allowed (Tables 3 and 4). Kabuli chickpeas should be sound, dry, fresh and cream to light brown in colour.

Failure to achieve the receival standards may mean price discounts, re-cleaning or, if severe, market rejection. [26]

26 Pulse Australia (2016) Receival and trading standards. Pulse Australia, http://pulseaus.com.au/marketing/receival-trading-standards

Table 3: *Example of visual charts designed to be used as a guide in conjunction with the current Australian pulse trading standards.*

Defect	Visual examples of defective chickpeas
Frost damaged, shrivelled and wrinkled	
Broken, chipped, loose seed coat, and split.	
Insect damaged, and sprouted.	
Hail damaged	

Source: Pulse Australia

12.6.1 Definitions

- *Defective grains:* a maximum of 2% field peas (in desi), 2% of grains that are poor-coloured grains, broken, damaged and split, shrivelled, distorted, grub eaten, sprouted, or affected by field mould.
- *Poor colour: a maximum of 2%* of cotyledon that is distinctly blemished and/or off-colour from the characteristic yellow colour of the predominate class, including a maximum of 1% visibly affected by Ascochyta blight.
- *Foreign material:* includes unmillable material and all foreign vegetable matter (i.e. anything that is not desi chickpeas, therefore includes cereals, oilseeds and other crops, and wild oats, and weed seeds not otherwise specified).
- *Unmillable material:* soil, stones, metal and other non-vegetable matter.

Table 4: *Receival standards for desi and kabuli chickpea.*

Chickpea type	Max. moisture content (%)	Min. purity (%)	Max. defective & poor colour	Screen size for defective seeds (mm)	Poor colour max. (%)	Foreign material max. in total (%)	Unmillable material max.	Snails max.	Insects max.
Desi	14	97	6	3.95 slotted	2% (but 1% Ascochyta)	3	0.5 (0.3% soil)	1 per 200 g	15 per 200 g
Kabuli	14	97	3	6 round	2% (but 1% Ascochyta)	3	0.5 (0.3% soil)	2 per 400 g	30 per 400 g

Source: Pulse Australia

Individual commodity traders are responsible for ensuring that specific country requirements and those pertaining to compliance with the Export Control Act 1982 are included as additional specifications on the contract.

MORE INFORMATION

http://www.pulseaus.com.au/storage/app/media/markets/2009_Chickpea-visual-quality.pdf

MORE INFORMATION

Manual of Importing Country Requirements

VIDEOS

1. Harvest weed seed control for the high rainfall zone.

2. Harvest – the time to get on top of resistant weeds.

3. A beginner's guide to harvest weed seed control.

MORE INFORMATION

IWM manual section on harvest weed management

12.7 Harvest weed-seed management

Controlling weeds after harvest may be more difficult in southern regions as there can be several months of good growing conditions for weeds.

In the southern cropping region's high rainfall zone (HRZ), an important question needs to be answered: how can harvest weed seed practices be adopted to reduce soil weed seed banks to address herbicide resistance? And more specifically, how can growers get weed seeds into the header?

Southern Farming Systems (SFS) is answering these questions through its Grains Research and Development Corporation-funded HRZ harvest weed seed control (HWSC) project. Paddock-scale trials will demonstrate to growers the suitability and effectiveness of a number of HWSC measures, using commercial equipment to highlight the potential of these management practices to complement large scale trials.

Trial plots have been established at SFS's Lake Bolac site in western Victoria, and in Tasmania. [27]

Trials in both south-eastern and western Australian grain-growing regions have found a 55 to 58 per cent reduction, overall, in the emergence of annual ryegrass across the three main harvest weed-seed control (HWSC) systems being practised by growers. [28]

12.7.1 Harvest weed seed control strategies

Weed seed capture and control at harvest can assist other tactics to put the weed seed bank into decline. Up to 95% of annual ryegrass seeds that enter the harvester exit in the chaff fraction. If it can be captured, it can be destroyed or removed.

Western Australian farmers and researchers have developed several systems to effectively reduce the return of annual ryegrass and wild radish seed into the seed-bank, and help put weed populations into decline.

A key strategy for all harvest weed seed control operations is to maximise the per cent of weed seeds that enter the header. This means harvesting as early as possible before weed seed is shed, and harvesting as low as is practical, e.g. 'beer can height'. Chickpea does not as readily lend itself to these practices of weed seed collection or confinement because of its late maturity relative to the weed seeds that have often fallen to the ground by crop harvest.

27 Watt S. (2016). Weed seed project aims to keep growers out of the woods. https://grdc.com.au/Media-Centre/Media-News/South/2016/03/Weed-seed-project-aims-to-keep-growers-out-of-the-woods

28 Clarry S. (2015). Trials measure harvest weed-seed control. https://grdc.com.au/Media-Centre/Ground-Cover/Ground-Cover-Issue-115-MarApr-2015/Trials-measure-harvest-weed-seed-control

Storage

Key messages

- Pulses stored above 12% moisture content (MC) require aeration cooling to maintain quality. The recommended air-flow rate for cooling grain is 2–3 litres of air per second per tonne of grain in the storage.
- Unlike cereal grains, pulses cannot be treated with protectants to prevent insect infestations. Therefore, meticulous hygiene and aeration cooling to manage storage temperature and moisture are crucial to preventing insect damage and moulds from downgrading stored chickpeas.
- Fumigation is the only option available to control pests in stored pulses. This requires a gas-tight, sealable storage.
- Avoiding mechanical damage to pulse seeds will maintain market quality and seed viability, and will make them less attractive to insect pests.
- It is important that growers minimise the number of handling operations wherever possible and use efficient handling techniques that minimise damage, for example belt conveyors rather than spiral augers.

The successful storage of pulses requires a balance between ideal harvest conditions and ideal storage conditions. Harvesting at 14% MC captures grain quality and reduces mechanical damage to the seed, but it also requires careful management of seeds in aerated silos to avoid deterioration during storage. [1]

Testa quality and physiological age are two principle components of chickpea, pea, lentil and faba bean seed quality. Both are affected by harvest and storage practices. Both also influence germination (although they are not the only factors) as well as other measures of seed quality which affect the ability of seeds to produce seedlings which can emerge and establish. [2]

Many of the quality characteristics of the grain from these crops are in the appearance, size and physical integrity of the seed. Mechanical seed damage, discolouration, disease, insect damage (Photo 1), split seeds or small seeds will lead to a downgrade in quality and market value. Buyers prefer large, consistently sized seed free of chemical residues for easy processing and marketing to consumers.

Photo 1: *Insect (cowpea weevil) damage on seeds of two 'kabuli' and three 'desi' chickpeas (from left to right). Photo: F. Erler* et al *99 (2009)*[3]

Unlike cereal grains, pulses cannot be treated with protectants to prevent insect infestations. Therefore, meticulous hygiene and aeration cooling to manage storage temperature and moisture are crucial to prevent insect damage and moulds from

1 P Burrill, P Botta, C Newman, B White, C Warrick (2014) Storing pulses. Fact sheet. Updated July 2014, https://grdc.com.au/Resources/Factsheets/2014/07/Grain-Storage-Fact-Sheet-Storing-Pulses

2 RH Ellis, PK Agrawal, EE Roos (1988) Harvesting and storage factors that affect seed quality in pea, lentil, faba bean and chickpea. In World Crops: Cool Season Food Legumes. Springer Netherlands, Dordrecht. 303–329.

3 Erler, F., Ceylan, F., Erdemir, T., & Toker, C. (2009). Preliminary results on evaluation of chickpea, *Cicer arietinum*, genotypes for resistance to the pulse beetle, Callosobruchus maculatus. Journal of Insect Science, 9(1), 58.

downgrading stored chickpeas. The Australian Pulse Standards stipulate standards for heat-damaged, bin-burnt, mouldy, caked or insect-infested chickpeas, and breaching of any of these can result in the discounting or rejection of product. [4] The effective management of stored chickpeas can eliminate all these risks. Growers contemplating medium–long-term storage (6–12 months) need to be aware that chickpeas continue to age, so that quality will deteriorate over time. Desi chickpeas will darken considerably in storage, with the rate of seed-coat darkening being accelerated by:

- high seed moisture content (MC)
- high temperatures
- high relative humidity

VIDEOS

1. GCTV Stored grain: Storing pulses.

Condition of the seed at harvest

- Seed subject to field weathering before harvest will deteriorate a lot quicker in storage, even when stored at acceptable temperature and relative humidity.
- Conditions of high relative humidity and high temperatures result in rapid deterioration in grain colour.
- To maintain yellow colour and minimise the darkening of seed, any grain stored >12% MC will require cooling and/or drying.
- Growers should avoid even short–medium storage of weather-damaged grain. [5]

Gaining a better understanding of the insect pests themselves, and fighting them using the right combination of management choices and equipment gives growers the upper hand. In a deregulated market there is a large range of domestic and export selling options. Growers strengthen their position when their storage facilities allow flexibility with grain handling and timing of sales.

As a bonus, many of the strategies used to minimise pest problems also significantly improve storage conditions for maintaining grain quality.

13.1 How to store product on-farm

Key points

- Combining good hygiene, well-managed aeration cooling and regular grain inspections provide the best foundation for successful grain storage.
- Findings of recent ecological research, which involved trapping flying storage pests across grain-growing regions, reinforced the value of cleaning up grain residues in storages and equipment.
- New, easy-to-use functions in automatic aeration controllers provide improved reliability of achieving good results from aeration cooling.
- Recirculation and ground-level applications have a role to play in effective, safe fumigation. [6]

13.1.1 Handling and storage of chickpea seed

Planting-seed selection

Special attention should be given to the harvest, handling and storage of planting seed retained on the farm. Seed should be:

- Sourced from the cleanest paddocks, where Ascochyta blight was not detected.
- Harvested at minimum of 13–14% MC to minimise mechanical damage to the seed.

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4 Pulse Australia (2011) Australian Pulse Standards, 2011/2012 Season. Pulse Australia, http://www.graintrade.org.au/sites/default/files/file/Commodity Standards/Pulse Standards 201112.pdf

5 Pulse Australia (2013) Northern Chickpea—Best Management Practices Training Course Manual 2013. Pulse Australia.

6 P Burrill (2013) Grain storage: future pest-control options and storage systems 2013–2014. GRDC Update Papers. GRDC, https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/07/Grain-Storage-Future-pest-control-options-and-storage-systems-2013-2014

- If heat dried, at temperatures below 40°C.
- Stored at approximately 13% MC.
- Kept at a grain temperature of below 30°C (Table 2).
- Graded to remove split, damaged and small seeds.

Handling

Grain may have been handled (augured) up to six times by the time it is delivered to receival points or is planted. It is important that growers minimise the number of handling operations, and use efficient handling techniques to minimise damage; e.g. using belt conveyors rather than spiral augers.

If using augers:

- Operate slowly and full.
- Use large-diameter augers.
- Length of the auger should be no longer than is necessary.
- Keep auger incline low.
- Check flight casing clearance—optimal clearance is typically 50% of grain size to minimise the grain being wedged between the auger spiral and the casing.

Seed longevity in storage

Growers contemplating medium- to long-term storage (6–12 months) need to be aware that chickpea seed continues to age, and that quality deteriorates over time (Table 1). Desi chickpea will darken considerably, and seed germination capacity and vigour will decline in storage, with the rate being accelerated by:

- high seed-moisture content
- high temperatures
- high relative humidity
- condition (weathering) of the seed at harvest.

Seed that has been weathered before harvest will deteriorate quicker in storage, even when stored under acceptable conditions of temperature and relative humidity. To maintain colour and minimise darkening of seed, any grain stored above 12% moisture will require cooling. [7]

Table 1: *Effect of moisture content and temperature on storage life of chickpea.*

Storage moisture (%)	Storage temperature (°C)	Longevity of seed (days)
12	20 30 40	>2000 500-650 110-130
15	20 30 40	700-850 180-210 30-50

Note: Most planting seed will need to be stored for a period of 180 days or more.

Source: Pulse Australia

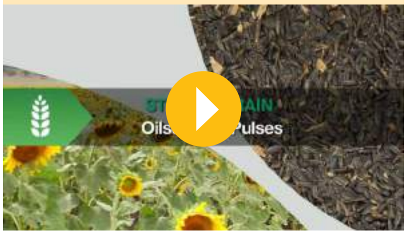
Moisture and temperature

Research has shown that harvesting pulses at higher moisture content (up to 14%) reduces field mould, mechanical damage to the seed, and splitting, and preserves seed viability. The challenge is to maintain this quality during storage, when there is an increased risk of deterioration at these moisture levels. As a result, pulses stored above 12% MC require aeration cooling to maintain quality.

7 B O'Mara, S Belfield, G Cumming (n.d.) Chickpea harvest and seed storage. Pulse Australia, http://www.pulseaus.com.au/storage/app/media/crops/2007_Chickpea-Harvest-Storage.pdf

VIDEOS

2. GCTV: Stored grain: Oilseeds and pulse storage



Grain Trade Australia (GTA) sets a maximum moisture limit of 14% for most pulses, but bulk handlers may have receival requirements as low as 12%. [8] As a general rule of thumb, the higher the moisture content, the lower the temperature required to maintain seed quality (Table 2). [9]

Table 2: *Maximum recommended storage period.*

		Grain temperature (°C)	
		20	**30**
Moisture content (%)	**14**	3 months	N/A
	13	9 months	3 months
	12	>9 months	9 months

Source: GRDC

Green pods and grains increase the risk of mould developing during storage—even at a lower moisture content. Aeration cooling will help prevent mould and hot spots by creating uniform conditions throughout the grain bulk. [10]

Weathering damage hinders storage

Pulses exposed to weathering before harvest deteriorate more quickly in storage. Chickpeas stored for the medium to long term (6–12 months) continue to age and lose quality (Table 1). Growers can minimise the effects of seed darkening, declining germination capacity and reduced seed vigour by:

- Lowering moisture content and temperature.
- Harvesting before weather damages the grain. [11]

13.1.2 On-farm storage

Growers in the southern region are investing in on-farm storage for a range of reasons. In the eastern states, on-farm storage gives growers options into domestic and export markets, while in South Australia—where the majority of grain goes to bulk handlers—growers tend to set up storage to improve harvest management.

Growers might only plan to store grain on-farm for a short time, but markets can change, so investing in gas-tight sealable structures means you can treat pests reliably and safely and leave your business open to a range of markets.

Growers should approach storage as they would purchasing machinery: Growers spend a lot of time researching a header purchase to make sure it is fit-for-purpose. Grain storage can also be a significant investment, and a permanent one, so it pays to have a plan that adds value to your enterprise into the future.

Agronomists tip: Decide what you want to achieve with storage, critique any existing infrastructure and be prepared for future changes: A good storage plan can remove a lot of stress at harvest – growers need a system that works so they capture a better return in their system. [12]

The two most common of the serious threats to grain quality in Australia's storages are insect pest infestations and grain moisture problems causing the growth of mould or fungus. Key initial strategies include:

8 Storing pulses. Stored Grain Information Hub. GRDC, www.grdc.com.au/GRDC-FS-GrainStorage-StoringPulses

9 B O'Mara, S Belfield, G Cumming (n.d.) Chickpea harvest and seed storage. Pulse Australia, http://www.pulseaus.com.au/storage/app/media/crops/2007_Chickpea-Harvest-Storage.pdf

10 Storing pulses. Stored Grain Information Hub. GRDC, http://storedgrain.com.au/storing-pulses/

11 GRDC Stored Grain Hub. Storing pulses. http://storedgrain.com.au/storing-pulses/

12 GRDC. (2015). Ground cover issue 119—Grain storage. Extension tailored for regional challenges. https://grdc.com.au/resources-and-publications/groundcover/ground-cover-issue-119-nov-dec-2015?f.Region%7Csregion=West&query=!allitems&meta_aissueno_not=%22Ground%20Cover%20supplements%22&personal=false&form=listing&collection=grdc-multi&profile=groundcover&meta_aissueno_not=GroundCover%E2%84%A2&meta_aissueno_not=%22Ground+Cover+supplements%22&matrix_origin=groundcover_details&meta_aissueno=119&fmo=on&start_rank=11

- Thorough hygiene maintained in storages and equipment.
- Keeping grain cool and dry grain in storage.

Attention paid to the three areas listed below will provide reliable grain quality:

- Good storage and equipment hygiene reduces early pest infestations and grain-contamination problems. Sieve and inspect grain in storages monthly.
- High-moisture grain in storage—have the right equipment and management strategies in place to deal promptly with any growth of mould or fungus. Monitor regularly.
- Cool grain temperature—use aeration to achieve cool, uniform temperature in storages in the first few weeks after harvest. Monitor to maintain these conditions. [13]

In most cases, for on-farm storage to be economical it will need to deliver on more than one of these benefits (Table 3). Under very favourable circumstances grain storage facilities can pay for themselves within a few years but it is also possible for an investment in on-farm storage to be very unprofitable. The grain storage cost-benefit analysis template is very useful step in the decision-making process to test the viability of grain storage on your farm. [14]

Table 3: *Advantages and disadvantages of grain storage options.*

Storage type	Advantages	Disadvantages
Gas-tight sealable silo	Gas-tight sealable status allows phosphine and controlled atmosphere options to control insects Easily aerated with fans Fabricated on-site or off-site and transported Capacity from 15 tonnes up to 3,000 tonnes Up to 25 year plus service life Simple in-loading and out-loading Easily administered hygiene (cone base particularly) Can be used multiple times in-season	Requires foundation to be constructed Relatively high initial investment required Seals must be regularly maintained Access requires safety equipment and infrastructure Requires an annual test to check gas-tight sealing
Non-sealed silo	Easily aerated with fans 7–10% cheaper than sealed silos Capacity from 15 tonnes up to 3,000 tonnes Up to 25 year plus service life Can be used multiple times in-season	Requires foundation to be constructed Silo cannot be used for fumigation—see phosphine label Insect control options limited to protectants in eastern states and Dryacide™ in WA Access requires safety equipment and infrastructure

13 P Burrill (2013) Grain storage: future pest-control options and storage systems 2013–2014. GRDC Update Papers. GRDC, https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/07/Grain-Storage-Future-pest-control-options-and-storage-systems-2013-2014

14 GRDC. (2015). Grain storage strategies in the northern region. https://grdc.com.au/Media-Centre/Hot-Topics/Grain-storage-strategies-in-the-northern-region

Storage type	Advantages	Disadvantages
Grain storage bags	Low initial cost Can be laid on a prepared pad in a paddock Provide harvest logistics support Can provide segregation options Are all ground operated Can accommodate high-yielding seasons Grain is untreated for wider market access.	Requires purchase or lease of loader and unloader Increased risk of damage beyond short-term storage (typically three months) Limited insect control options. Fumigation only possible under specific protocols Requires regular inspection and maintenance, which need to be budgeted for Aeration of grain in bags currently limited to research trials only Must be fenced off Prone to attack by mice, birds, foxes, etc. Limited wet weather access if stored in paddock Need to dispose of bag after use Single-use only
Grain storage sheds	Can be used for dual purposes 30 year plus service life Low cost per stored tonne	Aeration systems require specific design Risk of contamination from dual purpose use Difficult to seal for fumigation Vermin and bird control is difficult Limited insect control options without sealing Difficult to unload

Source: Kondinin Group

Established strategies

There is an increasing number of growers who are reaping rewards by placing extra emphasis on the management of their grain-storage facilities. It is the combination of practices listed below that provides the real strength to successful storage.

Key strategies include:

- High standard of hygiene for storages and grain-handling equipment—minimising insect-pest breeding sites.
- Monthly checks of grain in storage, including those of planting seed—sieving for insects and checking quality.
- Aeration—aeration fans fitted to storages, and operated by an automatic controller.
- Grain temperatures—checked and maintaining 20–23°C in summer and less than 15°C in winter.
- Fumigations, when required, are carried out in sealable silos. The silos are pressure-tested at least once a year.
- Storage record keeping—a simple system is used to record details such as grain variety, moisture content, any treatments given, inspection dates, and information on insects found and grain temperature.

MORE INFORMATION

Benefits flow from on-farm storage in the Mallee.

Grain storage facilities: Planning for efficiency and quality.

VIDEOS

3. Over the Fence: On-farm storage pays in wet harvest

New products and equipment

Aeration using automatic controllers

Reliability good results with aeration cooling are significantly increased with the use of automatic controllers to turn on silo fans when the best ambient temperature and humidity conditions are available. There are new functions in automatic aeration controllers. Some of the options available include:

- The ability to have fans automatically step through the three important stages of aeration cooling—continuous, purge, and protected.
- The ability to exclude very humid air (>85% RH) in all three of these stages.
- The ability to cater for fans with air-flow rates higher than the standard 2–3 litres per second per tonne (L/s/t).

These functions provide another good reason to stop using the less-reliable methods of trying to remember to manually switch fans on and off, or using power-point timers.

High–flow rotary grain cleaner

One of benefits of having storages on the farm is the ability to segregate different quality grades of grain at harvest time. For farmers who only just miss being given a premium grade due to a few extra percent screenings in wheat, or who face downgrades in pulses due to splits or weed seed contamination, grading is an option that can quickly add value.

Storage safety is also improved by grading out impurities and fines from oilseeds when filling silos.

The rotary grader has multiple screen tubes designed for flow rates that will keep up with most harvesting operations. A range of screen sizes and slot designs suit most grading requirements.

13.1.3 Silos

Well-designed and properly operated on-farm storage provides the best insurance that a grower can have to maintain the quality of chickpeas to be out-turned. Storages must be used in conjunction with sound management practices, which include monthly sieving for insects, regular grain-quality inspections, and ensuring that aeration cooling equipment is operating as required. [15]

Silos are the ideal storage option for pulses, especially if they are cone-based for easy out-loading with minimal seed damage (Photo 2). Because chickpeas are susceptible to splitting at the ideal storage moisture content of ≤12%, cone-based rather than flat-based silos are recommended for easy out-loading with minimal seed damage. For anything more than short-term storage (three months) aeration cooling and gas-tight sealable storage suitable for fumigation are essential features for best-practice quality control.

Always fill and empty silos from the centre holes. This is especially important with pulses because most have a high bulk density. Loading or out-loading off-centre will put uneven weight on the structure and may cause it to collapse. Avoid storing lentils in silos with horizontally corrugated walls, as the grain can run out from the bottom first and cause the collapse of the silo as, in bulk, the grain will slide down the silo walls rather than from the centre.

15 P Burrill, P Botta, C Newman, B White, C Warrick (2014) Storing pulses. Fact sheet. Updated July 2014, https://grdc.com.au/Resources/Factsheets/2014/07/Grain-Storage-Fact-Sheet-Storing-Pulses

Photo 2: *Sealable, aerated silos fitted with thermosiphons to assist with gas distribution during fumigation.*

Source: GRDC

Paint the outside of the silo with white paint. This reduces storage temperature by as much as 4–5 °C and can double the safe storage life of grains. Aerate silos with dry, ambient air. In addition to reducing storage temperatures, aeration is also effective in reducing moisture of seed harvested at high moisture content if flow rates are sufficient. Growers should avoid even short–medium storage of weather-damaged grain. [16]

Sealed silos offer a more permanent grain storage option than grain storage bags. Depending on the amount of storage required, they will have a higher initial capital cost than grain storage bags and are depreciated over a longer time frame than the machinery required for the grain bags. In a silo grain storage system as stored tonnage increases the capital cost of storage increases.

Potential advantages of using sealed grain silos as a method for grain storage include improved harvest management, reduced harvest stress, reduced harvest freight requirements, minimal insecticide exposure and the opportunity to segregate and blend grain.

Potential disadvantages of using sealed grain silos as a method for grain storage include the initial capital outlay, the outlay required to meet occupational health and safety requirements, the additional on farm handling required and the additional site maintenance requirements. [17]

16 DAF Queensland (2012) Chickpea: harvesting and storage. DAF Qld, https://www.daf.qld.gov.au/plants/field-crops-and-pastures/broadacre-field-crops/chickpeas/harvesting-and-storage

17 Francis J. (2006). An analysis of grain storage bags, sealed grain silos and warehousing for storing grain. https://grdc.com.au/uploads/documents/Final%20report%20Grain%20Storage%20Bags%2021%20Jul%20061.pdf

Pressure testing

At industry level, it is within growers' best interests to house grain in aerated, sealable storages to help curtail the rise of insect resistance to phosphine. Resistance has come about because of the prevalence of silos that are poorly sealed, or even unsealed, during fumigation. [18]

- A silo sold as a 'sealed silo' needs to be pressure tested to be sure it's gas-tight.
- It is strongly recommended that growers ask the manufacturer or reseller to quote the AS2628 on the invoice as a means of legal reference to the quality of the silo being paid for.
- Pressure test sealed silos upon erection, annually and before fumigating with a five-minute half-life pressure test.
- Maintenance is the key to ensuring a silo purchased as sealable can be sealed and gas-tight.

A silo is only truly sealed if it passes a five-minute half-life pressure test according to the Australian Standard AS2628. Often silos are sold as sealed but are not gas-tight — rendering them unsuitable for fumigation.

Even if a silo is sold as 'sealed' it is not sealed until it is proven gas-tight with a pressure test.

The term 'sealed' has been used loosely during the past and in fact some silos may not have been gas-tight from the day they were constructed.

However, even a silo that was gas-tight to the Australian Standard on construction will deteriorate over time so needs annual maintenance to remain gas-tight.

Why do I need to do a pressure test?

In order to kill grain pests at all stages of their life cycle (egg, larvae, pupae, adult), phosphine gas concentration levels need to reach and remain at 300 parts per million (ppm) for seven days or 200ppm for 10 days.

The importance of a gas-tight silo

Growers should pressure-test sealable silos once a year to check for damaged seals on openings. Storages must be able to be sealed properly to ensure high phosphine gas concentrations are held long enough to give an effective fumigation. At an industry level, it is in growers' best interests to only fumigate in gas-tight sealable storages to help stem the rise of insect resistance to phosphine. This resistance has come about because of the prevalence of storages that are poorly sealed or unsealed during fumigation. [19]

Research shows that fumigating in a storage that is not gas-tight does not achieve a sufficient concentration of fumigant for long enough to kill pests at all life cycle stages. For effective phosphine fumigation, a minimum gas concentration of 300 parts per million (ppm) for 7 days or 200 ppm for 10 days is required. Fumigation trials in silos with small leaks demonstrated that phosphine levels are as low as 3 ppm close to the leaks. The rest of the silo also suffers from reduced gas levels. [20]

It is recommended to pressure-test silos that are sealable once a year to check for damaged seals on openings. Storages must be able to be sealed properly to ensure effective fumigation.

There is no compulsory manufacturing standard for sealed silos in Australia. A voluntary industry standard was adopted in 2010. Watch this GRDC Ground Cover TV clip to find out more.

MORE INFORMATION

GRDC Pressure testing sealable silos factsheet.

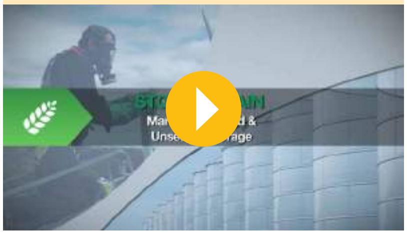
GRDC Silo buyer's guide

VIDEOS

4. Pressure testing sealed silos.

VIDEOS

5. Stored grain: Managing sealed and unsealed storage.



18 C Warrick (2012) Fumigating with phosphine, other fumigants and controlled atmospheres. Reprinted August 2012. GRDC, https://grdc.com.au/__data/assets/pdf_file/0025/206791/fumigating-with-phosphine.pdf.pdf

19 C Warrick (2011) Fumigating with phosphine, other fumigants and controlled atmospheres: Do it right—do it once: A Grains Industry Guide. GRDC Stored Grain Project, January 2011 (reprinted June 2013), https://grdc.com.au/__data/assets/pdf_file/0025/206791/fumigating-with-phosphine.pdf.pdf

20 P Botta, P Burrill, C Newman (2010) Pressure testing sealable silos. GRDC Grain Storage Fact Sheet, September 2010 Revised July 2014, http://storedgrain.com.au/wp-content/uploads/2014/09/GSFS-3_PressureTest-July14.pdf

MORE INFORMATION

Fumigating in a silo bag with phosphine?

VIDEOS

6. Grain bags best practice.

7. Extension Files: Grain bags—a grower's perspective.

13.1.4 Grain bags

Grain storage bags are relatively new technology offering a low cost alternative for temporary storage of grain to permanent grain storage structures on farm such as silos. Grain storage bags are made of multilayer polyethylene material similar to that used in silage fodder systems. Bags typically store between 200 and 220 tonnes of grain and are filled and emptied using specialised machinery (Photo 3). The bags are sealed after filling producing a relatively airtight environment which, under favourable storage conditions, protects grain from insect damage without the use of insecticides.

Potential advantages of using grain storage bags as a method for grain storage include the low capital set up costs, improved harvest management, less harvest stress, reduced harvest freight requirements, minimal cost in occupational health and safety (OH&S) requirements, reduced grain insecticide requirements and the opportunity to segregate and blend grain.

Potential disadvantages of using grain storage bags as a method for grain storage include the requirement for disposal of used bags, the period of storage before bag deterioration and the management necessary to ensure bag integrity. Another potential disadvantage of this system, when compared to permanent structures, is that once the storage period is complete there is no asset value in the storage system other than the bagging machinery. [21]

Photo 3: *A 100 m bag can be filled in 30 minutes with a constant supply of grain.*

Source: StarTribune.

Risks with chickpeas

Chickpeas can be stored successfully in silo bags of up to three months, but it is a less desirable option than silo storage. Marketers have rejected pulse grain because of moulds, taints and odours from storage in grain bags. Such taints and odours are not acceptable in pulse markets. [22]

Black discoloration of chickpeas due to moisture ingress into the base of grain bags has also occurred, causing serious losses in storage.

13.1.5 Grain storage—get the economics right

As growers continue to expand their on-farm grain storage, the question of economic viability gains significance. There are many examples of growers investing in on-farm grain storage and paying for it in one or two years because they struck the market at

21 Francis J. (2006). An analysis of grain storage bags, sealed grain silos and warehousing for storing grain. https://grdc.com.au/uploads/documents/Final%20report%20Grain%20Storage%20Bags%2021%20Jul%20061.pdf

22 W Hawthorne, A Meldrum, G Cumming (2010) Grain bags for pulse storage—use care. Australian Pulse Bulletin 2010 No. 3, http://www.pulseaus.com.au/storage/app/media/crops/2010_APB-Pulse-grain-bag-storage.pdf

the right time, but are these examples enough to justify greater expansion of on-farm grain storage?

The grain storage extension team conduct approximately 100 grower workshops every year, Australia wide and it's evident that no two growers use on-farm storage in the exact same way. Like many economic comparisons in farming, the viability of grain storage is different for each grower. Depending on the business's operating style, the location, the resources and the most limiting factor to increase profit; grain storage may or may not be the next best investment. For this reason, everyone needs to do a simple cost benefit analysis for their own operation.

Comparing on-farm grain storage

To make a sound financial decision, we need to compare the expected returns from grain storage versus expected returns from other farm business investments, such as more land, a chaser bin, a wider boomspray, a second truck or paying off debt. The other comparison is to determine if we can store grain on-farm cheaper than paying a bulk handler to store it for us.

Calculating the costs and benefits of on-farm storage will enable a return-on investment (ROI) figure, which can be compared with other investment choices and a total cost of storage to compare to the bulk handlers.

Cheapest form of storage

The key to a useful cost–benefit analysis is identifying which financial benefits to plan for and costing an appropriate storage to suit that plan. People often ask, "what's the cheapest form of storage?" The answer is the storage that suits the planned benefits. Short term storage for harvest logistics or freight advantages can be suited to grain bags or bunkers. If flexibility is required for longer term storage, gas-tight, sealable silos with aeration cooling allow quality control and insect control.

Benefits

To compare the benefits and costs in the same form, work everything out on a basis of dollars per tonne. On the benefit side, the majority of growers will require multiple financial gains for storing grain to make money out of it. These might include harvest logistics or timeliness, market premiums, freight savings or cleaning, blending, or drying grain to add value.

Costs

The costs of grain storage can be broken down into fixed and variable. The fixed costs are those that don't change from year to year and have to be covered over the life of the storage. Examples are depreciation and the opportunity or interest cost on the capital. The variable costs are all those that vary with the amount of grain stored and the length of time it's stored for. Interestingly, the costs of good hygiene, aeration cooling and monitoring are relatively low compared to the potential impact they can have on maintaining grain quality. One of the most significant variable cost, and one that is often overlooked is the opportunity cost of the stored grain. That is the cost of having grain in storage rather than having the money in the bank paying off an overdraft or a term loan.

The result

While it's difficult to put an exact dollar value on each of the potential benefits and costs, a calculated estimate will determine if it's worth a more thorough investigation. If we compare the investment of on-farm grain storage to other investments and the result is similar, then we can revisit the numbers and work on increasing their accuracy. If the return is not even in the ball park, we've potentially avoided a costly mistake. On the contrary, if after checking our numbers the return is favourable, we can proceed with the investment confidently.

Summary

Unlike a machinery purchase, grain storage is a long-term investment that cannot be easily changed or sold. Based on what the grain storage extension team are seeing around Australia, the growers who are taking a planned approach to on-farm grain storage and doing it well are being rewarded for it. Grain buyers are seeking out growers who have a well-designed storage system that can deliver insect free, quality grain without delay.

Table 4 is a tool that can be used to figure out the likely economic result of on-farm grain storage for each individual business. Each column can be used to compare various storage options including type of storage, length of time held or paying a bulk handler. [23]

Table 4: *Cost-benefit template for grain storage.*

Financial gains from storage		Example $/t
Harvest logistics/ timeliness	Grain price x reduction in value after damage % x probability of damage %	$16
Marketing	Post harvest grain price - harvest grain price	
Freight	Peak rate $/t - post harvest rate $/t	$20
Cleaning to improve grade	Clean grain price - original grain price - cleaning costs - shrinkage	
Blending to lift average grade	Blended price - ((low grade price x %mix) + (high grade price x %mix))	
Total benefits	Sum of benefits	$36.20
Capital cost	Infrastructure cost / storage capacity	$155
Fixed costs		
Annualised depreciation cost	Capital cost $/t / expected life storage eg 25yrs	$6.20
Opportunity cost on capital	Capital cost $/t x opportunity or interest rate eg 8% / 2	$6.20
Total fixed costs	Sum of fixed costs	$12.40
Variable costs		
Storage hygiene	(Labour rate $/hr x time to clean hrs / storage capacity) + structural treatment	$0.23
Aeration cooling	Indicatively 23c for the first 8 days then 18c per month / t	$0.91
Repairs and maintenance	Estimate e.g. capital cost $/t x 1%	$1.51
Inload/outload time and fuel	Labour rate $/hr / 60 minutes / auger rate t/m x 3	$0.88
Time to monitor and manage	Labour rate $/hr x total time to manage hrs / storage capacity	$0.24
Opportunity cost of stored grain	Grain price x opportunity interest rate e.g. 8% / 12 x No. months stored	$7.20
Insect treatment cost	Treatment cost $/t x No. of treatments	$0.35
Cost of bags or bunker trap	Price of bag / bag capacity tonne	
Total variable costs	Sum of variable costs	$11.32

MORE INFORMATION

GRDC Economics of on-farm grain storage, cost benefit analysis

Economics of on-farm grain storage: a grains industry guide

23 Warrick C. (2016). GRDC Update Papers: Grain storage—get the economics right. https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/09/Grain-storage-get-the-economics-right

VIDEOS

8. On-farm storage in the SA Mallee with Corey Blacksell.

9. On-farm storage in SA—Linden Price.

10. Over the Fence: On-farm storage delivers harvest flexibility and profit.

11. Stay safe around grain storage

Financial gains from storage		Example $/t
Total cost of storage	Total fixed costs + total variable costs	$23.72
Profit/Loss on storage	Total benefits - total costs of storage	$12.48
Return on investment	Profit or loss / capital cost x 100	8.1%

Source: GRDC.

13.2 Stored grain pests

Insects are not considered to be a major problem in stored chickpea. The exceptions appear to be where chickpeas have higher levels of splits and damaged seed, or are loaded into storages containing residues of cereal grains that are already infested with pests, or both. Then chickpeas may harbour several insects, including:

- rust-red flour beetle (*Tribolium castaneum*)
- lesser grain borer (*Rhyzopertha dominica*)
- saw-toothed grain beetle (*Oryzaephilus surinamensis*)

Where a prior infestation exists in storage facilities, pests can spread and develop in chickpeas. The most common pulse pests are the cowpea weevil and pea weevil (Note that pea weevil is a pest of field peas and infection occurs in the field. It is not a stored grain pest as such although adults can emerge from peas in storage) which can also survive and breed at slower rates in chickpeas. The cowpea weevil has a short life span of 10–12 days while the pea weevil only breeds one generation per year. [24] Hygiene is the most cost-effective method of managing bruchid problems. Growers need to thoroughly clean all residues of other pulses from headers, planting equipment, shed floors, augers and empty trucks and storages after each harvest, and whenever pulse seeds are handled on the farm. [25]

If weather damage before harvest or header settings have led to chickpeas containing higher levels of split grain and trash, they are more prone to infestation by pests such as the rust-red flower beetle. Pre-storage grading to remove splits or extra storage monitoring is required.

Chickpea gradings are attractive to storage pests. Gradings can act as a breeding site, causing infestations to spread to the storage complex. Use or remove gradings from the area as soon as possible.

Good hygiene by ensuring that all handling equipment and storages are clean before handling chickpea should prevent infestations from developing. If insects are found in stored chickpeas, the only registered treatment option are controlled atmospheres (CO_2, N_2) or phosphine fumigation. [26]

The high numbers of storage pests that can fly away from on-farm sources of infested grain in spring and summer looking for newly harvested clean grain to infest (Figure 1) demonstrates the value of regularly maintaining hygiene and inspecting grain.

24 Storing pulses. Stored Grain Information Hub. GRDC, http://storedgrain.com.au/storing-pulses/

25 DAF Queensland (2012) Chickpea: harvesting and storage. DAF Qld, https://www.daf.qld.gov.au/plants/field-crops-and-pastures/broadacre-field-crops/chickpeas/harvesting-and-storage

26 Pulse Australia. Chickpea harvest and seed storage. http://www.pulseaus.com.au/storage/app/media/crops/2007_Chickpea-Harvest-Storage.pdf

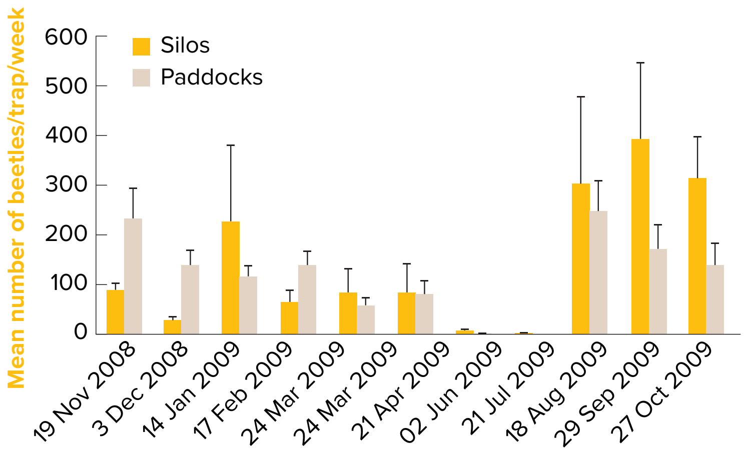


Figure 1: *Numbers of lesser grain borers trapped in warm and cooler months at farm silos and 1 km away in the paddocks.*

Source: GRDC

No insecticide sprays are currently registered for use on chickpea grain. Markets are particularly sensitive to insecticide residues, so the detection of any residues on chickpeas could result in the loss of a market, not just the rejection of a contaminated delivery. This is so whether the insecticide residue is from a storage application or from harvesting sooner than witholding periods (whp) allow.

For structural treatments of silos, use an inert dust such as diatomaceous earth (DE) after a thorough cleaning of all old grain residues. Wash out the silo with a pressure hose, then leave it open to dry is also recommended, particularly if an insect infestation occurred in the last grain stored in it. [27]

The aeration of pulses stored in silos is the key non-chemical tool used to minimise the risk of insect infestations and spoiling through heat and/or moisture damage (Figure 1). Pulses stored above 12% MC require aeration cooling to maintain quality. Australian pulse trading standards are set at a maximum moisture limit of 14% for chickpeas, and most other pulses [28], but bulk handlers may have receival requirements as low as 12%. As a general rule of thumb, the higher the moisture content, the lower the temperature required to maintain seed quality.

Aeration of chickpeas as soon as they go into the silo will provide uniform moisture conditions in the grain bulk, and quickly lower grain temperatures, which will minimise the effects of seed darkening, declining germination capacity and seed vigour.

13.2.1 Hygiene

Key points:

- Effective grain hygiene requires the complete removal of all waste grain from storages and equipment.
- Be meticulous with grain hygiene: pests only need a small amount of grain in order to survive and reproduce.
- Structural treatments, such as diatomaceous earth (DE), can be used on storages and equipment to protect against grain pests.
- Check delivery requirements before using chemical treatments, and avoid using with pulses and oil seeds.

27 Pulse Australia (2013) Northern Chickpea—Best Management Practices Training course Manual 2013. Pulse Australia.

28 Pulse Australia (2016) Australian Pulse Standards 2016–2017. Pulse Australia, http://www.pulseaus.com.au/storage/app/media/markets/20160801_Pulse-Standards.pdf

VIDEOS

12. GCTV2: Grain silo hygiene

The first line of defence against grain pests occurs before the pulses are put into storage—in meticulous grain hygiene. Because pest control options are limited, it's critical to remove pests from the storage site before harvest. The key to control is to ensure that all handling equipment and storages are cleaned of old grain residues before they are used to handle chickpeas. Effective hygiene plus aeration cooling can overcome 75% of pest problems in on-farm storage.

A bag of infested grain can produce more than one million insects during a year, and these can walk and fly to other grain storages where they will start new infestations.

Cleaning silos and storages thoroughly and removing spilt and leftover grain removes the feed source and harbour for insect pests.

Where to clean

Removing an environment for pests to live and breed in is the basis of grain hygiene, which includes all grain handling equipment and storages (Photo 4). Grain pests live in dark, sheltered areas, and breed best in warm conditions.

Common places where pests are found include:

- Empty silos and grain storages.
- Aeration ducts, augers and conveyers.
- Harvesters, field bins and chaser bins.
- Left-over bags of grain trucks.
- Spilt grain around grain storages.
- Equipment and rubbish around storages.
- Seed grain.
- Stockfeed grain. [29]

Successful grain hygiene involves cleaning all areas where grain gets trapped in storages and equipment. Grain pests can survive in a tiny amount of grain, so any fresh parcel of grain passing through machinery, storage or equipment can easily become infested.

Photo 4: *Grain left in trucks is an ideal place for grain pests to breed. Keep trucks, field bins and chaser bins clean.*

Source: Stored Grain Information Hub

29 Hygiene and structural treatments for grain storage. Fact sheet. Stored Grain Information Hub. GRDC, http://storedgrain.com.au/hygiene-structural-treatments/

When to clean

Straight after harvest is the best time to clean grain handling equipment and storages, before they have time to become infested with pests. A trial carried out at the start of a harvest in Queensland revealed more than 1,000 lesser grain borers in the first 40 L of grain through a harvester, which had been considered reasonably clean at the end of the previous season. Discarding the first few bags of grain at the start of the harvest is a good idea.

Studies have revealed that insects are least mobile during the colder months of the year. Cleaning around silos from July–August can reduce insect numbers before they become mobile.

How to clean

The better the cleaning job, the less chance there is of pests being harboured. The best ways to get rid of all grain residues use a combination of:

- sweeping
- vacuuming
- compressed air
- blow guns or vacuum guns
- pressure washers
- fire-fighting hoses

Using a broom or jets of compressed air gets rid of most grain residues, and a follow-up wash-down removes grain and dust left in crevices and hard-to-reach spots (Photo 5). Choose a warm, dry day to wash storages and equipment so they dry out quickly and do not get rusty. When inspecting empty storages, look for ways to make the structures easier to keep clean. Seal or fill any cracks and crevices to prevent grain lodging and insects harbouring. Bags of left-over grain lying around storages and in sheds create a perfect harbour and breeding ground for storage pests. After collecting spilt grain and residues, dispose of them well away from any grain storage areas.

Photo 5: *Clean silos, including the silo wall, with air and/or water to provide a residue-free surface to apply structural treatments.*

Source: Stored Grain Information Hub

The process of cleaning on-farm storages and handling equipment should start with the physical removal, blowing and/or hosing out of all residues. Once the structure is clean and dry, consider the application of DE as a structural treatment. (See Section 13.2.6 Structural treatments for chickpea storage for more information.)

Tip: A concrete slab underneath silos makes cleaning much easier (Photo 6).

Photo 6: *Concrete slab under silo makes cleaning up spilled grain much easier.*

Source: Stored Grain Information Hub

13.2.2 Aeration cooling

Key points:

- Grain temperatures below 20°C significantly reduce mould and insect development.
- Reducing grain temperature with aeration cooling protects seed viability.
- Controlling aeration cooling is a three-stage process: continual; rapid; and then maintenance.
- Stop aeration if the relative humidity of the ambient air exceeds 85%.
- Automatic grain-aeration controllers that select optimum fan running times provide the most reliable results.

Not all growers are convinced that the aeration of grain is worthwhile, or a valuable asset for their storage system in this region, and have been reluctant to install aeration fans in their storages. However, a well-managed aeration system typically reduces grain temperatures by at least 10°C (Figure 2). This has a significant impact on reducing insect-pest problems and in maintaining grain quality.

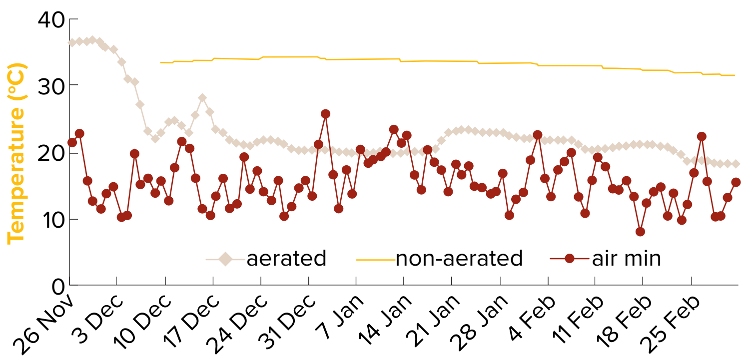


Figure 2: *Comparison of grain temperatures in aerated and non-aerated silos.*

Source: GRDC

Aeration cooling:

- Creates uniform conditions throughout the grain bulk.
- Prevents moisture migration.
- Maintains seed viability (germination and vigour).
- Reduces mould growth.
- Lengthens (and in some instances stops) insect reproduction cycles.
- Slows seed-coat darkening and quality loss.

Grain temperatures below 15°C stop the breeding cycle for all common storage pests. During summer, achieving grain temperature close to 20°C is also valuable, as it either stops or significantly slows insect population increases.

Fungal growth in grain in storage is kept in check with appropriate grain-moisture. Keeping grain at lower temperatures also assists to some extent in reducing fungal growth.

While adult insects can still survive at low temperatures, most of the young of storage pests stop developing at temperatures below 18–20°C (Table 5). At cool temperatures (20–23°C) insect pest life cycles (egg, larvae, pupae and adult) are lengthened from the typical four weeks at warm temperatures (30–35°C) to 12–17 weeks. [30]

Table 5: *The effect of grain temperature on insects and mould.*

Grain temp (°C)	Insect and mould development	Grain moisture content (%)
40–55	Seed damage occurs, reducing viability	
30–40	Mould and insects are prolific	>18
25–30	Mould and insects are active	13–18
20–25	Mould development is limited	10–13
18–20	Young insects stop developing	9
<15	Most insects stop reproducing, mould stops developing	<8

Source: Kondinin Group

With the support of an aeration controller, aeration can rapidly reduce stored grain temperatures to a level that helps maintain grain quality and inhibit insect development.

The recommended airflow rate for cooling grain is 2–3 litres of air per second per tonne (L/s/t) of grain in the storage.

Grain is an effective insulator because, like housing insulation, it holds many tiny pockets of air within a stack. Without aeration it will maintain its warm harvest temperature for a long time. Aeration cooling allows for longer-term storage of low-moisture grain by creating desirable conditions for the grain and undesirable conditions for mould and pests. Unlike aeration drying, aeration cooling can be achieved with air-flow rates of as little as 2–3 (L/s/t) of grain, from fans driven by a 0.37 kilowatt (0.5 horsepower) electric motor for silos of around 100 t capacity.

30 Aeration cooling for pest control. Stored Grain Information Hub. GRDC, http://storedgrain.com.au/aeration-cooling/; Keeping aeration under control (2010) Farming Ahead. No. 208, March 2010. Kondinin Group, http://storedgrain.com.au/wp-content/uploads/2013/06/Kondinin-Group-Report-Aeration-Controllers-Reduced.pdf

High-moisture grain can also be safely held for a short time with aeration cooling before it is blended or dried. Run fans continuously to prevent self-heating and quality damage. [31]

During trials where grain was harvested at 30°C and 15.5% MC, grain temperatures rose to 40°C within hours of being put into storage. An aeration controller was used to rapidly cool grain to 20°C and then hold the grain between 17–24°C from November to March.

Before replicating these results on the farm, growers need to:

- Know the capacity of their existing aeration system.
- Determine whether grain requires drying before cooling can be carried out.
- Understand the effects of relative humidity and temperature when aerating stored grain.
- Determine the target conditions for the stored grain.

MORE INFORMATION

Aeration cooling for pest control.

Air movement within the stack

The grain at the top of the stack is the hottest, as heat rises through the grain and the top grain is exposed to the head space in the silo (Figure 3).

As the air in the head space heats and cools each day, it creates ideal conditions for condensation to form. If this happens, the grain on the top of the stack will get wet.

Be aware that aeration drying requires specifically designed equipment and the process is much slower than aeration cooling or hot-air drying.

31 P Burrill, P Botta, C Newman, B White, C Warrick (2014) Storing pulses. Fact sheet. Updated July 2014, https://grdc.com.au/Resources/Factsheets/2014/07/Grain-Storage-Fact-Sheet-Storing-Pulses.

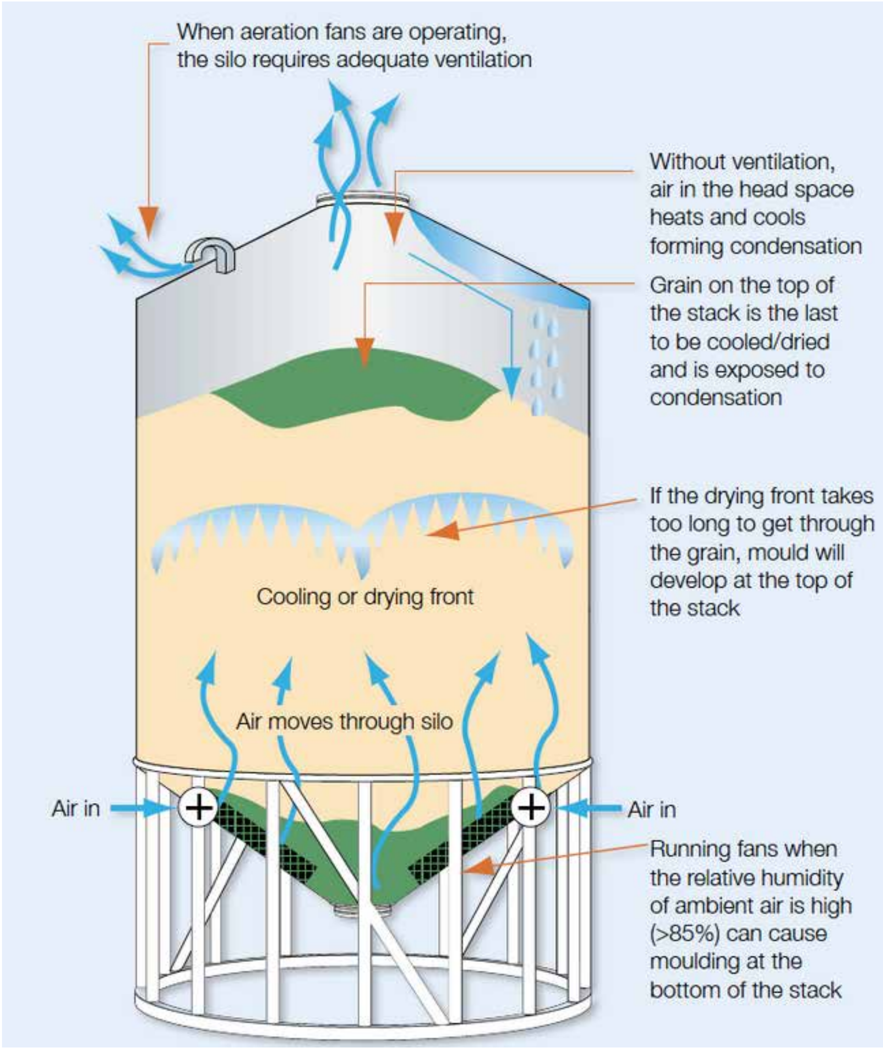


Figure 3: *Air movement within an aerated silo.*

Source: Stored Grain Information Hub

The cooling process

Operating an aeration fan for cooling requires a planned control program, which is best done with an automatic aeration controller. But even without a controller, growers need to aim for the same run time, following the same process.

The initial aim is to get maximum airflow through the grain bulk as soon as it enters storage, to stop it from sweating and heating.

When first loading grain into storage, run the aeration fans continuously from the time the grain covers the aeration ducts and for the next 1–3 days, until the cooling front reaches the top of the storage.

However, do not operate the aeration fans on continuous mode if the ambient relative humidity is higher than 85 per cent for extended periods of time, as this will wet the grain.

After the aeration fans have been running continuously for 2–3 days to flush out any warm, humid air, reduce the running time to 9–12 hours per day during the coolest part of the day, for the next seven days.

The goal is to quickly reduce the grain temperature from the mid-30s (°C) down to the low 20s (°C). An initial reduction in grain temperature of 10°C ensures grain is less prone to damage and insect attack, while further cooling becomes a more precise task. During this final stage, automated aeration controllers generally run fans during the coolest periods of the day, for an average of 100 hours per month.

Grain temperature is gradually reduced as low as possible and then maintained throughout the storage period.

Achieving reliable results with aeration cooling

Tips for producing the best results using aeration:

- Fan operations—replace manual switching or timers with automatic controllers to provide reliable, consistent cool grain temperatures. Buy them from a trustworthy supplier.
- Maintenance—manually test individual silo fans to check there are no electrical faults. For auto-controllers, check that the temperature and humidity sensor is clean. Also compare its readings with a reliable hand-held thermometer and RH (relative humidity) reader.
- Fan operations—ensure fans are stepped through the three important stages of aeration cooling. The most recent auto-controller models (e.g. Grainsafe-5000) automate this procedure over the first two weeks of storage.
- Fan performance—during 2011–2012 an Australian research group developed a simple, accurate method for testing the air-flow performance of aeration fans while they are operating. The recommended air-flow rate for cooling grain is 2–3 L/s/t of grain in the storage. Field tests on farm storages have shown that some fans do not deliver these rates. [32]

MORE INFORMATION

GRDC Performance testing aeration systems Fact sheet.

The risks of getting it wrong

Once in maintenance mode, running aeration fans on timers that are preset to run at the same time each day will not ensure the selection of the most appropriate air to maintain grain quality. The biggest risk with running aeration fans without a controller is forgetting to turn them off, or not being available to, if the relative humidity exceeds 85%.

Operating fans for extended periods of a few hours or days during humid conditions can increase grain moisture and cause moulding. Aeration controllers are designed to automatically select the best time to run aeration fans. Fans on these systems only run when the conditions will benefit the stored grain. [33]

Weevil development ceases at temperatures below 20°C. This is a strong incentive for aeration cooling, especially if gas-tight storage is not available. [34]

MORE INFORMATION

Aeration cooling for pest control.

VIDEOS

13. GCTV2: Grain storage cooling aeration.

Installation and management tips

When retrofitting an aeration system, avoid splitting airflow from one fan to more than one storage. Each storage will provide a different amount of back-pressure on the fan, resulting in uneven airflow and inefficient or even ineffective cooling.

If buying an aeration controller be aware that most controllers need to be installed by an electrician.

The preferred mounting location for aeration controllers is outside where the sensors can get ambient condition readings but are sheltered from the direct elements of the weather. To avoid the chance of a dust explosion, do not install aeration controllers in a confined space.

Ensure your electrician installs wiring that is properly insulated and protected from potentially damaging equipment, such as augers.

32 P Burrill (2013) Grain storage: future pest-control options and storage systems 2013–2014. GRDC Update Papers. GRDC, https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/07/Grain-Storage-Future-pest-control-options-and-storage-systems-2013-2014

33 Aeration cooling for pest control. Stored Grain Information Hub. GRDC, http://storedgrain.com.au/aeration-cooling/

34 Storing pulses. Stored Grain Information Hub. GRDC, www.grdc.com.au/GRDC-FS-GrainStorage-StoringPulses

Monitoring is a must

Aeration controllers reduce the amount of time operators need to physically monitor grain storages and turn fans on and off, but units and storage facilities still need to be checked regularly (Photo 7).

Photo 7: *Temperature check: monitor the effectiveness of the cooling process by checking grain temperature with a probe or a thermometer taped to a rod.*

Source: Stored Grain Information Hub

Most controllers have hour meters fitted, so run times can be checked to ensure they are within range of the expected total average hours per month.

Check fans to ensure they are connected and operating correctly.

The smell of the air leaving the storage is one of the most reliable indicators of whether the system is working. The exhaust air should change from a humid, warm smell to a fresh smell after the initial cooling front has passed through the grain.

Animals can damage power leads and automatic controller sensors and fan blades, or bearings can fail, so check these components regularly.

Check for suction in and feel for air-flow out of the storage vents when the fans are running.

Keeping grain at the right moisture and temperature levels will reduce the likelihood of insect infestations, but stored grain still needs to be sampled regularly and monitored for any changes. If possible, safely check the moisture and temperature of the grain at the bottom and top of the stack. [35]

13.2.3 Aeration drying

Ambient air can also be used to dry grain. Here, air is pumped through the grain bulk at high flow rates and at a temperature and humidity that will remove water from the grain (Table 6). [36]

Pulses stored for longer than three months at high moisture content (>14%) will require drying or blending to maintain seed quality. Aeration drying has a lower risk of cracking and damaging pulses, which occurs more readily with hot-air dryers. Unlike aeration cooling, drying requires high airflow rates of at least 15–25 L/s/t and careful management.

Aeration drying relies on a high air volume and is usually done in a purpose-built drying silo or a partly filled silo with high-capacity aeration fans. Aeration drying is a slow process and relies on four keys:

35 Aeration cooling for pest control. Stored Grain Information Hub. GRDC, http://storedgrain.com.au/aeration-cooling/

36 GRDC (2004) How aeration works. Grains Research Update Advice. GRDC, http://storedgrain.com.au/how-aeration-works-grdc-update/

- High airflow rates.
- Well-designed ducting for even airflow through the grain.
- Exhaust vents in the silo roof.
- Warm, dry weather conditions.

It is important to seek reliable advice on equipment requirements and correct management of fan run times, otherwise there is a high risk of damaging grain quality.

Management strategies

If ambient air conditions are such that grain will dry, fans should be turned on and left on. When conditions are such that grain is no longer drying, turn the fans off, and only turn them back on when air conditions will again allow grain to dry. The moisture content that grain will dry to is determined by the average condition of the air used. If the average condition of air used is too dry, grain below the drying front will be over-dried. To calculate if air of a certain quality will dry grain, training in calculating the equilibrium moisture content is needed. The data presented in Table 6 provides a rough guide. It should be noted that different types of grain have slightly different equilibrium grain moistures.

Table 6: *Approximate moisture content of grain resulting from aeration with air at various temperatures and humidity (equilibrium grain moisture content).*

Temp °C	Relative humidity (%)				
	30	40	50	60	70
15	9.8	11	12.1	13.4	15
25	9	10.3	11.4	12.8	14
35	8.5	9.7	10.7	12.0	13.5

Source: GRDC.

Automated controllers simplify the process of selecting air that is suitable for drying. If air conditions are such that drying will not occur, supplemental heating can be used to raise the air inlet temperature a few degrees. This greatly increases the potential of that air for drying, but care should be taken not to over dry. If supplementary heating is unavailable and the available air will not dry the grain, a short-term holding measure may be to change strategy and cool the grain, with the intent of maintaining its quality, until better quality air for drying is available. The grain is placed at risk of mould development if air conditions do not improve in the short term; and this risk is far greater if air of suitable quality to cool grain is also unavailable.

The best air for drying is often from midday to dusk, but this varies from region to region. There are 50 locations around Australia where the drying potential of air has been monitored and recorded over many years. This data can provide information on the frequency of air suitable to dry grain from varying moisture contents and will assist in determining if supplemental heating is likely to be needed. It also gives insight as to the time of the day when the best air quality for cooling or drying is likely to occur. [37]

High airflow for drying

Unlike aeration cooling, aeration drying requires high airflow, in excess of 15 L/s/t, to move drying fronts quickly through the whole grain profile and depth and carry moisture out of the grain bulk. As air passes through the grain, it collects moisture and forms a drying front. If airflow is too low, the drying front will take too long to reach the top of the grain stack – often referred to as a 'stalled drying front.' Providing the storage has sufficient aeration ducting, a drying front can pass through a shallow stack of grain much faster than a deep stack of grain. As air will take the path of least resistance, make sure the grain is spread out to an even depth.

37 GRDC (2004) How aeration works. Grains Research Update Advice. GRDC, http://storedgrain.com.au/how-aeration-works-grdc-update/

Ducting for drying

The way to avoid hot spots is with adequate ducting to deliver an evenly distributed flow of air through the entire grain stack (Photo 8). A flat-bottom silo with a full floor aeration plenum is ideal providing it can deliver at least 15 L/s/t of airflow. The silo may only be able to be part filled, which in many cases is better than trying to dry grain in a cone-bottom silo with insufficient ducting.

Photo 8: *Aeration drying requires careful management, high airflow rates, well designed ducting, exhaust vents and warm, dry weather conditions.*

Source: GRDC.

Venting for drying

Adequate ventilation maximises airflow and allows moisture to escape rather than forming condensation on the underside of the roof and wetting the grain on the top of the stack. The amount of moisture that has to escape with the exhaust air is 10 L for every one per cent moisture content removed per tonne of grain.

Weather conditions for drying

For moisture transfer to occur and drying to happen, air with a lower relative humidity than the grain's equilibrium moisture content must be used. For example, Table 7 shows that grain at 25°C and 14% moisture content has an equilibrium point of the air around it at 70% relative humidity. In order to dry this grain from its current state, the aeration drying fans would need to be turned on when the ambient air was below 70% relative humidity.

Phase one of drying

Aeration drying fans can be turned on as soon as the aeration ducting is covered with grain and left running continuously until the air coming out of the top of the storage has a clean fresh smell. The only time drying fans are to be turned off during this initial, continuous phase is if ambient air exceeds 85% relative humidity for more than a few hours.

Phase two of drying

By monitoring the temperature and moisture content of the grain in storage and referring to an equilibrium moisture table, a suitable relative humidity trigger point can be set. As the grain is dried down the equilibrium point will also fall, so the relative humidity trigger point will need to be reduced to dry down the grain further. Reducing the relative humidity trigger point slowly during phase two of the drying process will help keep the difference in grain moisture from the bottom to the top of the stack to a minimum, by ensuring the fans get adequate run time to push each drying front right through the grain stack.

Table 7: *Equilibrium moisture content for wheat. NOTE: values may be different for other grains.*

Relative humidity (%)	Temperature 15	25	35
	Grain moisture content (%)		
30	9.8	9.0	8.5
40	11.0	10.3	9.7
50	12.1	11.4	10.7
60	13.4	12.8	12.0
70	15.0	14.0	13.5

Source: GRDC.

Supplementary heating

Heat can be added to aeration drying in proportion to the airflow rate. Higher airflow rates allow more heat to be added as it will push each drying front through the storage quick enough to avoid over heating the grain close to the aeration ducting. As a general guide, inlet air shouldn't exceed 35°C to avoid over heating grain closest to the aeration ducting.

Cooling after drying

Regardless of whether supplementary heat is added to the aeration drying process or not, the grain should be cooled immediately after it has been dried to the desired level. [38]

13.2.4 Cooling or drying: making a choice

It can be difficult to know whether grain needs to be dried or cooled, but there are some simple rules of thumb to help you decide. For longer-term storage grain must be lowered to the correct moisture content. Grain that is dry enough to meet the specifications for sale can be cooled, without drying, to slow insect development and maintain quality. Grain of moderate moisture can be either cooled for short periods to slow mould and insect development, or dried providing the right equipment and conditions are available. After drying to the required moisture content, grain can be cooled to maintain quality. High-moisture grain (for example, 16% or more for wheat) will require immediate moisture reduction before cooling for maintenance. [39]

13.2.5 Aeration controllers

Aeration controllers manage both aeration drying, cooling and maintenance functions in up to ten separate storages (Photo 9). The unit it takes into account the moisture content and temperature of grain at loading, the desired grain condition after time in storage and selects air accordingly to achieve safe storage levels.

A single controller has had the ability to control the diverse functions of aeration: cooling, drying and maintenance. The controller can not only combine the ability to control all three functions, but automatically selects the correct type of aeration strategy to obtain the desired grain moisture and temperature. [40]

MORE INFORMATION

Dealing with high moisture grain.

VIDEOS

14. Aeration drying – getting it right.

MORE INFORMATION

Aerating stored grain: cooling or drying for quality control.

38 GRDC Stored Grain Information Hub: Dealing with high moisture grain. http://storedgrain.com.au/dealing-with-high-moisture-grain/

39 Aeration cooling for pest control. Stored Grain Information Hub. GRDC, http://storedgrain.com.au/aeration-cooling/

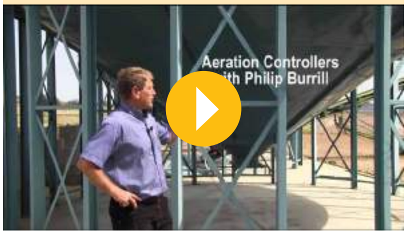
40 GRDC. (2007). Ground Cover Issue 57 – New Generation in aeration controller. https://grdc.com.au/Media-Centre/Ground-Cover-Supplements/Ground-Cover-Issue-57-Grain-Storage-Supplement/New-generation-aeration-controller

MORE INFORMATION

Performance testing aeration systems.

VIDEOS

15. Aeration controllers with Philip Burrill.



Research has shown that with the support of an aeration controller, aeration can rapidly reduce stored grain temperatures to a level that helps maintain grain quality and inhibits insect development.

During trials where grain was harvested at 30°C and 15.5% moisture, grain temperatures rose to 40°C within hours of being put into storage.

An aeration controller was used to rapidly cool grain to 20°C and then hold the grain between 17–24°C during November through to March.

Before replicating similar results on farm, growers need to:

- Know the capacity of their existing aeration system.
- Determine whether grain requires drying before cooling can be carried out.
- Understand the effects of relative humidity and temperature when aerating stored grain.
- Determine the target conditions for the stored grain.

Photo 9: *Automatic aeration controllers are the most effective way to cool grain and are designed to manage many storages, from one central control unit.*

Source: GRDC.

13.2.6 Structural treatments for chickpea storage

Chemical sprays are not registered for pulses in any state in Australia. While there is a maximum residue limit (MRL) for dichlorvos on lentils, the product is only registered for use on cereal grains.

Chemicals used for structural treatments do not list the specific use before storing pulses on their labels, and MRLs in pulses for those products are either extremely low or nil. Using chemicals even as structural treatments risks exceeding the MRL, so is not recommended.

One possibility of a structural treatment is using diatomaceous earth (DE), an amorphous silica that is sold commercially as Dryacide®. It acts by absorbing the insect's cuticle or protective waxy exterior, causing death by desiccation. Before applying DE for use with pulses, wash and dry the storage and all equipment to be used in the application to remove any residues left from previous years. This will ensure the DE doesn't discolour the grain surface. If applied correctly, with complete coverage in a dry environment, DE can provide up to 12 months of protection for storages and equipment. [41]

If unsure, check with the grain buyer before using any product that will come into contact with the stored grain.

41 Storing pulses. Stored Grain Information Hub. GRDC, http://storedgrain.com.au/storing-pulses/

Application

Inert dust requires a moving airstream to direct it onto the surface being treated; alternatively, it can be mixed into a slurry with water and sprayed onto surface. Follow the label directions. Throwing dust into silos by hand will not achieve an even coverage, and so will not be effective. For very small grain silos and bins, a hand-operated duster, such as a bellows duster, is suitable. Larger silos and storages require a powered duster operated by compressed air or a fan. If compressed air is available, it is the most economical and suitable option for use on the farm; connect it to a Venturi duster (e.g. Blovac BV-22 gun) (Photo 10).

Photo 10: *A blower/vacuum or Venturi gun are the best applicators for inert dusts. Aim for an event coat of diatomaceous earth across the roof, walls and base.*

Photo: C. Warrick, Proadvice

The application rate is calculated at 2 g/m^2 of the surface area treated. Although DE is inert, breathing in excessive amounts of it is not ideal, so use a disposable dust mask and goggles during application (Table 8).

Silo application

Apply inert dust in silos, starting at the top (if safe), by coating the inside of the roof then working your way down the silo walls, finishing by pointing the stream at the bottom of the silo. If silos are fitted with aeration systems, distribute the inert dust into the ducting without getting it into the motor, where it could cause damage. [42]

Table 8: *Diatomaceous earth application guide.*

Storage capacity (t)	Dust quantity (kg)
20	0.12
56	0.25
112	0.42
224	0.60
450	1.00
900	1.70
1,800	2.60

MORE INFORMATION

Hygiene and structural treatments for grain storages Fact sheet.

VIDEOS

16. Applying diatomaceous earth demonstration

42 Pulse Australia (2013) Northern Chickpea—Best Management Practices Training Course Manual 2013. Pulse Australia.

13.2.7 Fumigation

The only control options against stored pests are phosphine, an alternative fumigant, or a controlled atmosphere.

Protectant insecticide sprays, as commonly used to protect cereal grains against insect infestations, cannot be used with pulses. Phosphine is the only fumigant currently registered for use in pulses (Photo 11), and successful fumigation requires a storage that can be sealed gas-tight.

Photo 11: *Phosphine is widely accepted as having no residue issues.*

Photo: DAF Qld

There is some resistance to phosphine, the grain industry has adopted a voluntary strategy to manage the build-up of phosphine resistance in pests. Its core recommendations are to limit the number of conventional phosphine fumigations on undisturbed grain to three per year, and to employ a break strategy. Phosphine is widely accepted as causing no residue problems. [43]

Phosphine application

You achieve effective fumigation by placing phosphine tablets at the rate directed on the label onto a tray and hanging the tray in the top of a pressure-tested, sealed silo, or into a ground-level application system if the silo is fitted with recirculation. Keep the silo sealed for 7–10 days: seven days if the grain temperature is above 25°C, and 10 days if it is 15–25°C. Do not fumigate if the temperature inside the silo is less than 15°C as insects will not be active and the phosphine will therefore be ineffective. After the waiting period, open the top lid of the silo and ventilate grain for a minimum of one day with aeration fans running, or five days if no fans are fitted. A minimum withholding period of two days is required after ventilation before grain can be used for human consumption or stock feed. The total time required for fumigating ranges from 10–17 days. Read label directions.

When using phosphine, it is important that gas concentrations are held at high levels for the full fumigation exposure time. Immature stages of the insects and resistant strains that are being found more frequently will be controlled by phosphine only in a sealed, gas-tight storage. Phosphine is toxic to people as well as insects, so do not

43 P Collins (2009) Strategy to manage resistance to phosphine in the Australian grain industry. Cooperative Research Centre for National Plant Biosecurity, http://www.graintrade.org.au/sites/default/files/file/NWPGP/Phosphine%20Resistance%20Strategy.pdf

handle treated grain before the completion of the 7–10-day exposure period and the required airing period.

Fumigating in silo bags

In some situations, growers may find it difficult to gain access to a sealable, gas-tight silo in which to carry out effective fumigation of infested grain. Trials have shown that a silo bag can be used successfully for fumigation with phosphine when using the correct procedure.

Key steps:

- A gas-tight seal—inspect the silo bag and repair any minor holes.
- Correct phosphine tablet dose—apply in multiple grain spears evenly placed 7 m apart
- Allow a 14-day fumigation period.
- Vent the gas safely using a standard F650 aeration fan.

High concentrations of phosphine can be maintained for the required length of time to fumigate grain successfully in a silo bag. Fumigation trials on a standard 75-m-long bag containing ~230 t of grain were successful in controlling all life stages of the lesser grain borer.

When using phosphine in silo bags, remember that it is illegal to mix phosphine tablets with grain because of residue issues. Separate them by using perforated conduit to contain tablets and spent dust. The 1-m tubes can be speared horizontally into the silo bag and removed at the end of the fumigation. Trial results suggest that the spears should be no more than 7 m apart and fumigation should occur over 12–14 days. In previous trials when spears were spaced 12 m apart, the phosphine diffused through the grain too slowly. [44]

Fumigating in silos

The standard recommended practice for phosphine application has been to place tablets in trays in the sealed silo headspace. For small- and medium-sized silos (i.e. <150 t capacity) this is an effective method. The phosphine gas only takes approximately 24 hours to diffuse from the top to the bottom, through the 5–7 m depth of grain to the base of the silo.

For larger, taller silos (>150 tonnes), however, it can take two or more days for phosphine gas to reach the grain in the base of the silo. This can be a problem for a standard 7 or 10-day fumigation period, because any infested grain at the bottom does not get enough exposure to high gas concentrations to kill all stages of the insects.

The answer is recirculation. This is simply a system of adding plumbing to the silo to connect the silo base with the top of the silo to speed up the movement of gas through all grain in it. Recirculation should provide faster, more uniform gas distribution.

A number of silo manufactures now offer silos fitted with PVC tubes that run down the outside of silos from top to bottom. The complete system, including the silo itself, still needs to be well sealed so that it is gas-tight. Otherwise gas will leak out and the fumigation will fail to kill all the pests.

The two main systems are:

- Recirculation—has silo roof-to-base plumbing, with the addition of a small aeration fan that is used to force the phosphine gas around the silo. The critical time to have this operating is during the first 3–4 days, while tablets are liberating gas.
- Thermosiphon—has the same plumbing arrangement as the recirculation system, without the use of an electric fan. The heating and cooling of the silo head space

44 Silo bag fumigation (2012) Northern Update, Issue 66, Spring 2012. GRDC, http://www.icanrural.com.au/newsletters/NL66.pdf

and the black piping down side of the silo (passive heat exchange) is used to help generate air currents which distribute the phosphine gas.

A useful piece of equipment is a ground-level application box. To assist with safety and reduce the amount of time climbing silos to place tablets, phosphine tablets or bag chains can be placed in appropriate structures or containers on the ground at the base of the silo. These are often part of a recirculation system and connect to the internal aeration ducting.

Whatever system is used, it is very important to ensure it is designed so that it has ample space for tablets with free gas flow, to prevent the phosphine gas concentrations building up above the flammable limit, above 17,000 ppm, when it becomes explosive. Do not restrict gas movement with small containers for tablets or small-diameter pipe while gas is liberated in the first 3–4 days. Seek advice to ensure only safe designs are used. [45]

Non-chemical treatment options

Non-chemical treatments include:

- Carbon dioxide—treatment with CO_2 involves displacing the oxygen inside a gas-tight silo with CO_2, which creates a toxic atmosphere to grain pests. To achieve a complete kill of all the main grain pests at all life stages, CO_2 must be retained at a minimum concentration of 35% for 15 days.
- Nitrogen—grain stored under N_2 also provides insect control and quality preservation without chemicals. It is safe to use, and environmentally acceptable, and the main operating cost is the capital cost of equipment and electricity. It also leaves no residues, so grains can be traded at any time, unlike with chemical fumigants, which have withholding periods. Insect control with N_2 involves using pressure-swing adsorption (PSA) technology, and works by modifying the atmosphere inside the grain storage to remove everything except N_2, thereby starving the pests of oxygen. [46]

The carbon dioxide and nitrogen methods are sometimes referred to as a controlled atmosphere, because the composition of air in the silo is changed. They are more expensive than using phosphine, but they offer an alternative for resistant pest species.

MORE INFORMATION

Fumigating with phosphine, other fumigants and controlled atmospheres.

13.3 Monitoring stored chickpeas

Like cereal grains, chickpeas need to be delivered with nil live storage insects. [47] It is essential that any insect pests present in the on-farm storage are identified so that the best use of both chemical and non-chemical control measures can be exploited to control them.

Growers are advised to monitor all grain storages every two weeks during warmer periods of the year and at least monthly during cool periods (Photo 12). Use sieving and quality inspections to monitor stored pulses, and keep records of what you find. Use one of the GRDC publications on stored pest identification to help. Also record any fumigation action taken. If safe, visually check, smell and sample grain at the bottom and top of the stack regularly. [48] Having sample ports fitted in the side of the silos also enables temperature probe checks and grain sampling.

MORE INFORMATION

GRDC's Stored grain pest identification: The back pocket guide

45 P Burrill (2013) Grain storage: future pest-control options and storage systems 2013–2014. GRDC Update Papers. GRDC, https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/07/Grain-Storage-Future-pest-control-options-and-storage-systems-2013-2014

46 C Warrick (2011) Fumigating with phosphine, other fumigants and controlled atmospheres. GRDC, https://grdc.com.au/__data/assets/pdf_file/0025/206791/fumigating-with-phosphine.pdf.pdf

47 Pulse Australia (2013) Northern Chickpea—Best Management Practices Training Course Manual 2013. Pulse Australia.

48 P Burrill, P Botta, C Newman (2010) Aeration cooling for pest control. Fact sheet. GRDC, http://storedgrain.com.au/aeration-cooling/

Photo 12: *Keep records of findings from monitoring insects in stored grain.*

Source: DAF Qld

Here are some basic points to follow when monitoring for insect pests in your pulses:

- Sample and sieve grain from the top and bottom of grain storages for early pest detection. Probe or pitfall traps placed into the top of the grain will often detect storage-pest insects before you can see them in your sieve, as the traps remain in the grain all the time.
- Holding an insect sieve in the sunlight will encourage insect movement, making pests easier to see. Sieve samples onto a white tray, again to make small insects easier to see. Sieves should be of 2 mm mesh and need to hold at least 0.5 L of grain.
- One way to help identify live grain pests is to place them into a glass container and hold them in sunlight to warm the grain and insects. This will encourage activity without overheating or killing them. Rice weevils, cowpea bruchids and saw-toothed grain beetles can walk up the walls of the glass easily, but flour beetles and lesser grain borers cannot. Look closely at the insects walking up the glass. Rice weevils have a curved snout, saw-toothed grain beetles do not; and cowpea bruchids have a globular, tear-shaped body. [49]

13.4 Grain protectants for storage

No protectants are registered for use on pulses and oilseeds.

49 GRDC (2011) Stored grain pests identification: The back pocket guide. GRDC, https://grdc.com.au/resources-and-publications/all-publications/publications/2016/09/grdc-bpg-storedgrainpests

Environmental issues

Key messages

- Environmental stresses during seed development have a negative effect on the quality of chickpea seeds.
- Freezing temperatures at the late vegetative stage can cause considerable damage and yield losses.
- Chickpeas are prone to waterlogging, and as there are no in-crop control measures to deal with waterlogging, a critical management tool is avoidance of high-risk paddocks.
- Both low and high temperatures can limit the growth and grain yield of chickpea at all phenological stages. Temperature is a major environmental factor that regulates the timing of flowering, and thus influences grain yield.
- After disease, the major constraint to greater chickpea production is its sensitivity to the end-of-season (terminal) drought that occurs in both the Mediterranean-type climates and when grown on stored soil moisture in the summer-rainfall region of Australia. Unlike many other crops, chickpea is unable to escape terminal drought through rapid development because low temperatures (<16°C) often cause flower and pod abortion.
- Chickpeas are extremely sensitive to salinity, and can have difficulty accessing water and nutrients from saline layers in the soil.
- Chickpeas are classified among the most sensitive of all field crops to sodic soil conditions.

14.1 Frost issues for chickpeas

Radiant can be frost is a major stress to crops, and one of the principal limiting factors for agricultural production worldwide, including Australia. Radiant frosts occur when plants and soil absorb sunlight during the day and radiate heat during the night when the sky is clear and the air is still. Dense, chilled air settles into the lowest areas of the canopy, where the most serious frost damage occurs. The cold air causes nucleation of the intracellular fluid in plant tissues, and this causes the plasma membrane to rupture. [1]

Legumes, including chickpeas, field peas, faba beans and lentils, are very sensitive to chilling and freezing temperatures, particularly at the stages of flowering, early pod formation and seed filling, although damage may occur at any stage of development.

Frosts (or isolated freezing events) are a problem for chickpeas (Photo 1) in southern Australia, especially when they occur in the late vegetative and reproductive phenological (climate-induced developmental) stages, and the air temperature drops to 2°C or less on clear nights in early spring. They occur most frequently after the passing of a cold front, when the moisture and wind dissipates, leaving cold and still conditions with clear skies.

1 A Maqbool, S Shafiq, L Lake (2010) Radiant frost tolerance in pulse crops: a review. Euphytica 172 (1), 1–12.

VIDEOS

1. GCTV: Extreme temperature analysis to better understand frost events.

2. GCTV3: Frost R&D.

3. Frost's emotional impact, is it greater than its economic impact?

Photo 1: *Frost damage to a chickpea crop.*

Source: ABC Rural 2013

Areas of high frost risk in southern Australia include the Eyre Peninsula, Murray Mallee and the mid-north of South Australia, and the Wimmera Mallee region of Victoria. Over the last few years, the worst-affected areas have had crop production losses close to 100%. [2]

The occurrence and extent of frost damage tends to be affected by the microclimate, with great variability occurring within paddocks and even on the same plant. Therefore, soil type, soil moisture, position in the landscape, and crop density can have a bearing on the damage caused by a frost. In some species, crop nutrition has been shown to mitigate the effect of freezing range temperatures on the plant. It is thought that fertilisation of the plant, and consequent fast growth rates, can exacerbate the effect of freezing, particularly on the part of the plant undergoing elongation.

14.1.1 Industry costs

Crop losses due to frost are estimated to average more than $33 million a year in SA and Victoria, and over the whole of Australia may cost the grains industry on average more than $100 million a year. [3] The real cost of frost is a combination of the monetary cost due to both reduced yield and quality, and the hidden cost of management tactics used to minimise frost risk. These include:

- delaying sowing and its associated yield reduction
- sowing less profitable crops such as barley and oats
- avoiding cropping on the valley floors, which are among the most productive parts of the landscape.

14.1.2 Impacts on chickpea

Chickpea seedlings are tolerant of frost; however, the plants have low tolerance to frost during the flowering stage due to the exposed nature of flowers. Isolated frost events during the reproductive stage commonly results in flower or pod abortion, and this can be detrimental to yield in environments that experience terminal drought.

2 M Rebbeck, G Knell. (2007) Managing frost risk: a guide for southern Australian growers. SARDI, GRDC.

3 M Rebbeck , G Knell (2007) Managing frost risk: a guide for southern Australian growers. SARDI, GRDC.

As well as causing the rupture of the plasma membranes of cells, frosts can also cause the dehydration of cells as a result of the freezing of the extracellular spaces. [4]

Different varieties of chickpeas and other pulses have different levels of tolerance to frost, allowing farmers to choose the varieties best suited to conditions in their district (Table 1).

Table 1: *Chickpea varieties differ in their frost susceptibility and risk exposure times.*

Frost tolerance of pods & seeds	Commencement of flowering	Duration of flowering	Example variety	How frost is tolerated or avoided
Low	Early	Medium	Genesis™079	Vulnerable to early frosts and flowering under cool conditions
Low	Medium	Medium	Genesis™ 090	Avoids early frosts, but forces flowering to occur in heat
Low	Late	Short–medium	Almaz$^{\phi}$, Nafice$^{\phi}$	Avoids early frosts, but forces flowering to occur in heat

Source: Pulse Australia

MORE INFORMATION

Pulse Australia (2015) Minimising frost damage in pulses. Australian Pulse Bulletin. Updated 20 November.

Damage to vegetative growth

Damage is more likely to occur where the crop has grown rapidly during a period of warm weather and is then subjected to freezing temperatures. In chickpeas, the elongation regions are often the first affected by freezing, and this can show up in a frost-damaged plant by sigmoidal curves around the elongation point: this is commonly referred to as 'hockey stick' (Photo 2). Depending on the minimum temperature and the duration of the frost, plants may be partially damaged, resulting in lower yield and quality at harvest or even complete crop failure; or they may be killed outright.

Sub-zero temperatures in winter and spring (late frosts) can damage the leaves and stems of the plant. Frosts can cause bleaching of the leaves, especially on the margins. However, chickpeas have an excellent ability to recover from this superficial damage, and are able to regenerate new branches in severe cases. Late frosts also cause flower, pod and seed abortion.

4 A Maqbool, S Shafiq, L Lake (2010) Radiant frost tolerance in pulse crops: a review. Euphytica 172 (1), 1–12.

Photo 2: *Frost can cause bends like a hockey stick in chickpea stems.*

Photo S. Loss, DAFWA

The effect is readily visible, and may be seen as patches in the field, or on individual plants or branches of plants. Damage is usually more severe where stubble has been retained. Regrowth will generally occur provided soil moisture levels are adequate.

Chickpeas may be able to recover sufficiently to flower and set pods following an isolated frost event during the reproductive growth stage, provided soil moisture conditions are favourable during the subsequent periods.

In the field, frost tolerance decreases from the vegetative stage to reproductive stage. [5]

Damage to flowers and pods

Freezing temperatures damage leaves and destroy flowers and developing seeds (Photos 3 and 4). The time and duration of flowering affects tolerance and the ability to compensate after the frost. Early flowers are often aborted in chickpeas, but if soil moisture is available long-duration cultivars can compensate for the loss. Frosts that occur toward the end of the reproductive period following podset are more damaging, resulting in the abortion of pods and large yield reduction. [6]

Frost will normally affect the earliest-formed pods low on the primary and secondary branches. (By contrast, pod abortion induced by moisture stress is normally noted on the last-formed pods at the tips of the branches.) Pods at a later stage of development are generally more resistant to frost than flowers and small pods, but may suffer some mottled darkening of the seed coat. Provided soil moisture is adequate, varieties with an extended podding period can compensate for damage better than varieties that tend to pod up over a shorter period.

Minimum temperatures <5°C during the reproductive stage will kill the crop, but new regrowth can occur from the base of the almost-killed plants if moisture conditions are favourable. [7]

Frost is most damaging to yield:

- when it occurs during later flowering and early pod fill

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5 A Maqbool, S Shafiq, L Lake (2010) Radiant frost tolerance in pulse crops: a review. Euphytica 172 (1), 1–12.

6 JS Croser, HJ Clarke, KHM Siddique, TN Khan (2003) Low-temperature stress: implications for chickpea (*Cicer arietinum* L.) improvement. Critical Reviews in Plant Sciences 22 (2), 185–219, https://www.researchgate.net/publication/234520461_Low-Temperature_Stress_Implications_for_Chickpea_Cicer_arietinum_L_Improvement

7 Pulse Australia (2013) Northern Chickpea—Best Management Practices Training Course: Manual 2013. Pulse Australia.

- under dry conditions where moisture limits the plant's ability to re-flower and compensate for frost damage

Photo 3: *Frost damage to leaves.*

Photo: G. Cumming, Pulse Australia

Photo 4: *Chickpea frosted at flowering.*

Source: Pulse Australia

14.1.3 Managing to lower frost risk

The different conditions under which the frost occurs will influence what management practices will be more effective. The options are to:

- Delay flowering.
- Avoid high inputs.
- Sow more frost-tolerant crops and pastures.
- Grow hay.
- Avoid sowing susceptible crops in frost-prone areas, such as low lying places.
- Sow and graze dual-purpose crops.
- Encourage cold-air drainage. Consult a specialist.
- Add clay to paddocks with sandy surfaces.

Frost risk is difficult to manage in pulses, however some management strategies may reduce the risk or the extent of damage. These include:

- Knowing the topography, and map areas of greatest risk so that they can be managed to minimise frost damage.
- Choosing the right crop type, crop variety and sowing time to reduce exposure or impact at vulnerable growth stages.
- Carefully assessing the soil type, condition, and soil-moisture levels, and managing stubble and the crop canopy.
- Correcting crop nutrition and minimising stressors of the crop to influence the degree of frost damage.

Ensure crops have an adequate supply of trace elements and macro-nutrients. Crops deficient or marginal in potassium and copper are likely to be more susceptible to frost damage; this may also be the case for molybdenum. [8]

Problem areas and timings

Mapping or marking areas identified as frost-prone will enable growers to target frost and crop management strategies to these high-risk areas.

Knowing when the period of greatest probability of frost occurs is also important for crop management.

Crop and sowing time

The main strategy used to minimise frost risk in broadacre cropping has been to sow crops later. Risks exist with delayed sowing, even though this practice can reduce the probability of crops flowering in a frost-risk period. Crops sown later can still be affected by frost.

Strategies to minimise frost damage in pulses work in combinations of:

- growing a more tolerant species
- trying to avoid having peak flowering and early podding during the period of most risk
- extended flowering to compensate for losses to frost
- ensuring that most grain is sufficiently filled to avoid damage when frost occurs (Table 1).

Targeting flowering and early podding to periods of the lowest probability of frost is achieved through combinations of sowing date and variety choice based on flowering time and flowering duration. Local experience will indicate the best choices.

By planting for late flowering, farmers target the avoidance of early frosts, but in the absence of frost, late flowering may reduce yields if moisture is deficient or there are high temperatures.

Very early flowering can allow pods to be sufficiently developed to escape frost damage, and ensure some grain yield at least before a frost occurs. Increased disease risk needs to be considered with early sowing.

Spread the risk

Match different pulses to risk areas by sowing a different variety or species into targeted areas within the same paddock. Matching the crop, variety, sowing date and subsequent inputs to the frost-risk location spreads the risk.

Have forage as an optional use. Designating hay or forage as a possible use for the pulse in paddocks with a high frost risk provides flexibility.

Mixing two pulse varieties (e.g. long and short season, tall and short) balances the risks of frost and of end-of-season (terminal) drought, and reduces the risk of losses from any one-frost event. Multiple frost events can damage both varieties. If grain

8 Pulse Australia (2015) Minimising frost damage in pulses. Australian Pulse Bulletin. Updated 20 November, http://pulseaus.com.au/growing-pulses/publications/minimise-frost-damage

from both varieties is not of the same delivery grade, then only the lowest grade is achieved. The only realistic, practical options are in peas, narrow-leafed lupins, kabuli chickpeas; perhaps desi chickpeas are an option. Differences in flowering times are minimal in lentils and beans.

Sowing a mixture of pulse species is feasible, but not common. Complications in crop choice include achieving contrasting grain sizes, herbicide requirements, harvest timing and grain cleaning. Multiple frosts may damage both crops. Pulses grown in a mix will be suitable for feed markets only unless they can be cleaned to enable purity in segregation. If these difficulties can be overcome there is an opportunity for alternate-row sowing of different pulses.

Reduce frost damage

Managing inputs. To minimise financial risk in frost-prone paddocks when growing susceptible crops, growers can:

- Apply conservative rates of fertilisers to frost-prone parts of the landscape.
- Avoid using high sowing rates.

Advantages of avoiding high inputs are:

- Less financial loss if the crop is badly frosted.
- Lower-input crops, though potentially lower yielding during favourable seasons, are less like to suffer severe frost damage than higher-input crops with a denser canopy.
- Input costs saved on the higher frost-risk paddocks may be invested in other areas where frost risk is lower.

Lower sowing rates may result in a less dense canopy that increases crop tillering and may allow more heating of the ground during the day, and transfer of this heat to the canopy at night. However, there is no hard evidence that lower sowing rates will reduce frost damage.

The main disadvantage of this practice is that in the absence of frost, lower grain yield and/or protein may be the result during favourable seasons, contributing to the hidden cost of frost. (This is a particular disadvantage in barley and wheat delivery grades.) Less-vigorous crops can also result in the crop being less competitive with weeds.

Managing nodulation and nutrition. Ensure pulse crops are adequately nodulated and fixing nitrogen. Ensure pulses have an adequate supply of trace elements and macronutrients, although supplying high levels is unlikely to increase frost tolerance. Crops deficient or marginal in potassium and copper are likely to be more susceptible to frost damage, and this may also be the case for molybdenum. Foliar application of copper, zinc or manganese may assist, but only if the crop is deficient in the element applied.

Managing the canopy. A bulky crop canopy and exposure of the upper pods may increase frost damage to pulses. Semi-leafless, erect peas may be more vulnerable than conventional, lodging types because their pods are more exposed. A mix of two varieties of differing height, maturity and erectness may also assist in reducing frost damage.

Sow in wider rows, so that frost is allowed to get to ground level, and the inter-row soil is more exposed. An open canopy does not trap cold air. Wide rows require the soil to be moist to trap the heat in the soil during the day. With wide or paired rows and a wide gap, the heat can radiate up, however this may not always be effective.

Channel cold air flow away from the susceptible crop by using wide rows aligned up and down the hill or slope. Where cold air settles, a sacrifice area may be required.

The presence of cereal stubble makes the soil cooler in the root zone, worsening the frost effect compared with bare soil. Standing stubble is considered less harmful than slashed stubble as less light is reflected and the soil is more exposed to the sun. Dark-coloured stubble will be more beneficial than light-coloured types.

Rolling can help keep soils warm by slowing soil-moisture loss, but not necessarily on self-mulching or cracking soils. Note that press wheels roll only in the seed row, and not the inter-row. With no-till practice, avoid having bare, firm, moist soil as it will lose some of its stored heat.

Claying or delving sandy soils increases the ability of the soil to absorb and hold heat by making the soil colour darker, and retaining moisture nearer the surface.

Higher carbohydrate levels in the plant during frost leads to less leakage during thawing. A higher sugar content (high Brix) will also have a lower freezing point, and associated protection against frost damage. The effectiveness of various products applied to soil and plants to increase plant carbohydrates is unknown.

Better varieties coming. Through Pulse Breeding Australia, the GRDC is investing in germ plasm enhancement and variety breeding to increase frost tolerance in pulses. The focus is on altered flowering time and duration to avoid frost, and screening of pulse varieties for relative levels of frost tolerance in the field. New varieties will be released when available. [9]

A five-year research project funded by GRDC examined the effects of agronomic practices on frost risk in broadacre agriculture in southern Australia. The researchers manipulate the soil heat bank to store heat during the day and release heat into the canopy of the crop at night. The research examined how the crop canopy could be manipulated to allow for warm air from the soil to rise and increase the temperature at crop head height (Figure 1). They have identified strategies that could be used to significantly reduce the impact of frost.

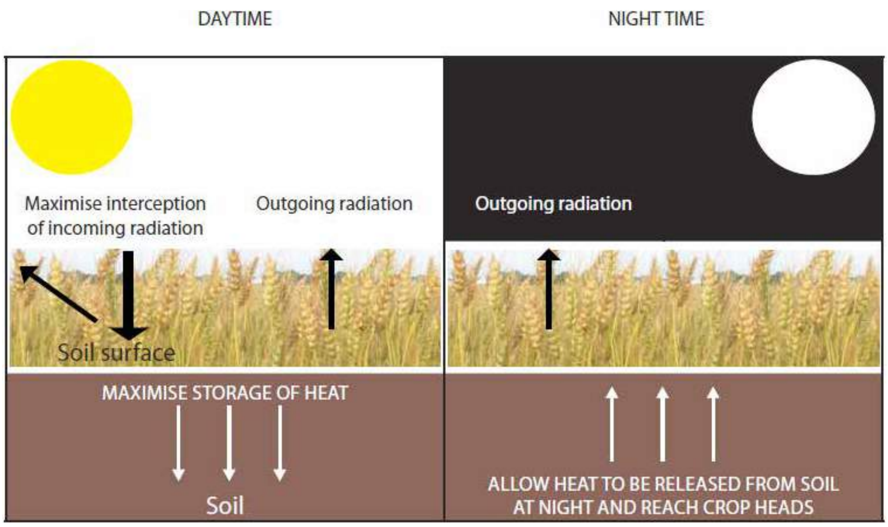


Figure 1: *Temperature dynamics in a crop canopy and canopy interactions.*

Source: Rebbeck and Knell 2007

Importance of soil moisture

Soil moisture is the most important factor for storing soil heat that will be released to and through the crop canopy at night. Because water has a high specific heat, radiation cooling overnight will be reduced when moisture is present in the soil. On a daily basis, heat is transferred into and out of approximately the top 300 mm of soil. When the soil is wet, heat transfer and storage in the upper soil layer is higher, so more heat is stored during daytime for release during the night.

There is also some evidence that moist soils can retain their warming properties for more than 24 hours, allowing some scope for an accumulation of heat from sunlight for more than one day. Heavier textured soils hold more moisture (and therefore heat) than lighter textured soils. A more dense soil can hold more moisture within the

9 Pulse Australia (2015) Minimising frost damage in pulses. Australian Pulse Bulletin. Updated 20 November, http://pulseaus.com.au/growing-pulses/publications/minimise-frost-damage

soil surface for heat absorption and subsequent release. Darker soils also absorb more light energy than lighter soils. Water-repellent sandy soils are usually drier at the surface than normal soils, and are therefore more frost prone. Frost studies in SA have found that crops were likely to be more damaged on lighter soil types because the soil temperature is lower as a result of lower soil moisture and the more reflective nature of these soils. On such soils, clay spreading or delving may be an option for reducing frost risk.

Use of agronomic practices

Table 2 shows the rankings of agronomic practices, adopted in both SA and WA, in order of importance. The table shows the paddock management strategies that manipulate the soil heat bank or manipulate the canopy air flow within the paddock, followed by paddock management strategies that also may assist crops to better tolerate frost. The final column in the table shows the reduction in frost damage from adopting these various practices in frost prone regions (derived from project trials).

The frost avoidance strategies, described in Table 2, are whole farm approaches to reduce or spread risks of frost injury. [10]

Table 2: *Agronomic practices to reduce frost risk ranked in order of importance.*

Soil heat bank manipulation ranking	Description	Increased temp at canopy height (ave) (°C)	Reduction in frost damage
Clay delving or clay spreading	In soils with a sandy surface, clay delving increases heat storage, nutrient availability and infiltration rate. Reducing frost risk by increasing the clay content of sandy-surfaced soils is the strongest finding in South Australia.	1.0	Up to 80%
Rolling	Rolling sandy soil and loamy clay soil after seeding has reduced frost damage, although the results were not statistically significant.	0.5	Up to 18%
Removing stubble	Removing stubble had a negligible effect on yield and frost risk. The role stubble plays in retaining soil moisture could be more important.	0.5	Minimal
Manipulation of the crop canopy ranking	**Description**	**Increased temp at canopy height (ave) (°C)**	**Reduction in frost damage**
Blending varieties and variety selection	Blending long- and short-season wheat varieties is a way to hedge your bets against frost or end-of-season drought within a paddock. A similar risk profile occurs when sowing one paddock with each variety at the same time. Successful results have been achieved in SA and WA blending Krichauff or Wyalkatchem with Yitpi. Certain varieties, such as Yitpi, Stiletto and Camm, flower later. Long-season varieties frequently avoid frost by flowering later in the growing season, when frost incidence is less. To further reduce frost risk, these varieties should be sown towards the middle or end of a wheat-sowing program rather than first.	0.0	Yitpi 12% less damaged than Krichauff
Cross-sowing	Crops sown twice with half the seed sown in each run gives an even plant density and has been found to more slowly release the soil heat, so that it can have an impact on air temperature at head height in early morning when frosts are most severe. This practice will incur an increased sowing cost. This result is based on two trials in WA.	0.6	13%

10 M Rebbeck, G Knell (2007) Managing frost risk: a guide for southern Australian growers. SARDI, GRDC.

Soil heat bank manipulation ranking	Description	Increased temp at canopy height (ave) (°C)	Reduction in frost damage
Wide row spacing	Wide-row sowing (e.g. 230–460 mm spacings) were ineffective for reducing frost damage. Wide-row crops consistently yield 10–15% less than the standard sowings with or without frost. In the presence of minor or severe frost, frost damage was similar for normal and wide row spacings.	0.2	0%
Lower sowing rate	A lower sowing rate (35–50 kg/ha) on frost-prone paddocks has not yet been proven to minimise frost damage. In WA, the plants in thinner crops appear more robust and able to better withstand frost events. The extra tillers formed per plant spread flowering time over a longer window. However, the crop is less competitive with weeds.	0.0	0%

Source: Rebbeck and Knell 2007

VIDEOS

4. Managing the effects of frost.

14.1.4 Managing frost affected crops

There are a number of options available for managing crops that have been frosted (Table 3). The following table highlights these options and the pros and cons of each. The suitability of each option will depend on the severity of the frost and analysis of costs versus returns. [11]

Table 3: *Options to manage frosted crops.*

Option	Advantages	Disadvantages
Harvest	No damage estimates required Salvage remaining grain Condition stubble for seeding	Costs may be greater than returns Need to implement weed control Threshing problems Need to remove organic matter
Hay and silage*	Stubble removed Weed control	Cost per hectare Quality may be poor (especially in wheat)
Chain or rake	Retains some stubble and reduces erosion risk Allows better stubble handling	Cost per hectare Time taken
Graze	Feed value Weed control	Inadequate stock to utilise feed (see Figure 6) Remaining grain may cause acidosis Stubble may be difficult to sow into
Spray	Stops weed seedset Preserves feed quality for grazing Gives time for decisions Retains feed Retains organic matter	Difficulty getting chemicals onto all of the weeds with a thick crop May not be as effective as burning Boom height limitation Cost per hectare Some grain still in crop

11 DAFWA (2016) Frost and cropping, https://www.agric.wa.gov.au/frost/frost-and-cropping

Option	Advantages	Disadvantages
Plough	Recycles nutrients and retains organic matter Stops weed seedset Green manure effect	Requires offset discs to cut straw Soil moisture needed for breakdown and incorporation of stubble
Swath	Stops weed seedset Window can be baled Regrowth can be grazed Weed regrowth can be sprayed	Relocation of nutrients to windrow Low market value for straw Poor weed control under swath Cost per hectare
Burn	Recycles some nutrients Controls surface weed seeds Permits re-cropping with disease control Can be done after rain	Potential soil and nutrient losses Fire hazard Organic matter loss

Source: DAFWA

* Less likely option with chickpea.

Photo 5: *Frosted pulses make excellent quality forage, although chickpea may be less so.*

Source: Pulse Australia.

14.2 Waterlogging and flooding issues

Waterlogging is probably one of the most important factors limiting the growth of crops and pasture in the high rainfall regions (>500 mm per annum) of southern Australia. [12] Chickpeas are prone to waterlogging (Photo 6), and as there are no in-crop control measures to deal with this problem, a critical management tool is the avoidance of high-risk paddocks, based on previous experience and paddock history.

12 K Moore, M Ryley, M Sharman, J van Leur, L Jenkins, R Brill (2013) Developing a plan for chickpeas 2013. GRDC Update Papers. February 2013, http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/02/Developing-a-plan-for-chickpeas-2013

Photo 6: *Water logging in chickpeas.*

Source: Australian UAV

14.2.1 Symptoms

Chlorosis has been observed after four days of waterlogging, firstly on the upper leaves. Reddish-brown anthocyanin pigmentation also develop on midribs, stems and some leaflets. Leaflets fold upward, a symptom typical of moisture stress. Unexpanded leaves have necrotic margins. Abscission (shedding) of chlorotic leaflets can begin six days after waterlogging and will progress until most of the plant is defoliated. [13]

Symptoms of waterlogging can be confused with those of Phytophthora root rot but differ as follows (Table 4):

- Plants are most susceptible to waterlogging at flowering and early pod-fill.
- Symptoms develop within two days of flooding, compared to at least seven days for Phytophthora root rot.
- Roots are not rotted, and at first are not easily pulled from the soil.
- Plants often die too quickly for the lower leaves to drop off.

Table 4: *Differences between Phytophthora root rot and waterlogging.*

Phytophthora root rot	Waterlogging
Organism kills roots	Insufficient oxygen kills roots
Chickpeas, medics, lucerne are hosts	No link with cropping history or weed control
Occurs at any time of year	Usually occurs later in the year
Symptoms begin after a week or more	Symptom begin within two days
Lower leaves often yellow and fall off	Plants die too fast for leaves to yellow or fall
Roots always rotted and discoloured	Initially roots not rotted or discoloured (tips black)
Plants easily pulled up and out	Plants not easily pulled up initially

13 AL Cowie, RS Jessop, DA MacLeod (1989) Effect of waterlogging on photosynthesis and stomatal conductance of chickpea leaves. 5th Australian Agronomy Conference, University of New England.

Effect of waterlogging on stomatal conductance and photosynthesis

Stomatal conductance is the measure of the rate of passage of carbon dioxide (CO_2) entering, or water vapour exiting through the stomata of a leaf, and photosynthesis is a process that converts light energy into chemical energy that can fuel growth. Within 24 hours, stomatal conductance of waterlogged chickpeas can decline, and will completely stop within three days. One day after waterlogging, photosynthesis and stomatal conductance have been recorded at 87% and 36%, respectively, of unaffected plants. Rapid decline in stomatal conductance over 24 hours, followed by a sharp decrease in photosynthesis between two and four days, suggest that waterlogging decreases photosynthesis through stomatal closure. Stomatal closure may also be caused by a decrease in potassium uptake, or production of abscisic acid or ethylene by the plant. Reduction in photosynthesis may result from the effects of waterlogging on carboxylation enzymes and the loss of chlorophyll, in addition to the effect of stomatal closure. [14]

IN FOCUS

How the timing of waterlogging affects chickpeas

The effect of the timing of waterlogging on chickpeas was examined in two pot trials. Plants were waterlogged for 10 days at different stages of growth: from 21 days after sowing (DAS); at flowering or at mid-pod fill; and combinations of these times.

Waterlogging at any stage reduced seed yield; waterlogging at 21 DAS had the least effect, reducing yield relative to the non-waterlogged control by 35%.

The ability of the plants to survive and regrow following waterlogging decreased with increasing age: mortality rate averaged 0% after waterlogging at 21 DAS, 30% when it occurred at flowering, and 100% at pod fill (Table 5). Tolerance to waterlogging was not enhanced by previous exposure to waterlogging.

In a second experiment, waterlogging was imposed at six different times shortly before or after flowering began. Ability to survive waterlogging declined sharply as flowering commenced: the mortality rate increased from 13% when waterlogging was imposed six days before flowering, to 65% one day after flowering, and 100% when waterlogging began 7.5 days after flowering (Table 5). it is suggested that survival and recovery after waterlogging may have been inhibited in flowering plants by an inadequate supply of nitrogen or carbohydrates. [15]

14 AL Cowie, RS Jessop, DA MacLeod (1989) Effect of waterlogging on photosynthesis and stomatal conductance of chickpea leaves. 5th Australian Agronomy Conference, University of New England.

15 AL Cowie, RS Jessop, DA MacLeod (1996) Effects of waterlogging on chickpeas. I. Influence of timing of waterlogging. Plant and Soil 183 (1), 97–103, http://www.regional.org.au/au/asa/1992/poster/soil-stress/p-04.htm

Table 5: *Mortality rate and regrowth following waterlogging imposed at five stages of floral development. Means followed by the same letter are not significantly different.*

Planting times	1	2	3	4	5
Time of waterlogging (days after sowing)	66	61	56	51	46
Time of flowering (days after waterlogging)	-7.5	-6	-1	+2	+6
Mortality rate (%)	100a	94a	63b	38c	13d
Total regrowth per pot (mg dry weight)	0a	0.4ab	18.1bc	32.6c	50.6d

Source: Cowie *et al.* 1996

14.2.2 Management options for waterlogging

- Avoid poorly drained paddocks and those prone to waterlogging.
- Do not flood-irrigate after podding has commenced, especially if the crop has been stressed.

A rule of thumb is that if the crop has started podding and the soil has cracked do not irrigate. Overhead irrigation is less likely to result in waterlogging, but consult your agronomist before proceeding. [16]

Innovative management techniques to reduce waterlogging

Waterlogging should be seen as a major threat to a farmer's potential income. The losses attributed to waterlogging in Tasmania, especially in high-rainfall years, run into the millions of dollars. What management techniques can be used to mitigate these losses and how can they be justified financially?

Before making any management decisions to reduce waterlogging, it is beneficial to understand where the problem areas lie, the size of these areas, how often losses occur in them, and how much potential earnings might increase if waterlogging is significantly reduced.

A monitoring program can be implemented to analyse all of the available data. These can include, but are not limited to, the following:

- Soil aspects—type, health, depth, characteristics, organic matter, electrical conductivity, compaction, topography, capacity to hold available water.
- Water—soil moisture, depth of water table, entry point (e.g. next door, over-irrigation, rainfall), direction of movement (on surface or sub-surface), exit points, irrigation management.
- Crop analysis – visual inspections (by ground or air), vigour mapping (NDVI), health/sap checks, yield mapping.

Once all the data have been collected, informed decisions can be formed to make management changes to reduce losses.

There are many tools available to reduce crop losses from waterlogging; some are listed below.

16 K Moore, M Ryley, M Schwinghamer, G Cumming, L Jenkins (2015) Chickpea: Phytophthora root rot management. Australian Pulse Bulletin. Updated 20 November 2015. Pulse Australia, http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/phytophthora-root-rot

Management systems:

- controlled traffic farming
- minimum tillage, no tillage, conservation or strip tillage
- sub-soiling
- crop selection
- managing livestock to reduce compaction
- surveying and planning for drainage, including watershed simulation

Drainage:

- Surface drainage:
- shallow field drains
- large excavated ditches
- land forming
- open excavated drains
- raised beds
- hump and hollow
- Sub-surface drainage:
- agpipe or tile installation
- mole ploughing
- gravel mole ploughing

Irrigation management:

- machinery selection—choosing the right pivot nozzles for your soil type
- irrigation scheduling
- control systems
- variable rate irrigation (VRI)
- salinity considerations

After making changes, it is vitally important to continue monitoring to gauge improvement and to prompt further action. This system helps ensure that there is a reduction in crop losses attributed to waterlogging. As it is far easier to select the low-hanging fruit first, it is recommended that you choose the easiest option first and then progress to the harder projects. [17]

VIDEOS

5. GCTV3: Big wet: Managing strategies after flooding.

14.3 Temperature

Both low and high temperatures can limit the growth and grain yield of chickpeas at all phenological stages. Temperature is a major environmental factor regulating the timing of flowering, thus influencing grain yield. The production of the cool-season chickpea is constrained by low temperatures across much of its geographical range, including southern Australia. Cold stress generally occurs in the late vegetative and reproductive stages across the geographical areas of chickpea production. Cold and freezing temperatures (–1.5°C to 15°C) are considered to be a major problem during the seedling stage of winter-sown chickpea in Mediterranean-climate areas and of autumn-sown crops in temperate regions. Southern Australia is most affected by chilling temperatures at flowering. On the other hand, high day and night temperatures (>30 during the day, and >16°C at night) may cause damage during the reproductive stage on winter-sown chickpeas in Mediterranean-type rainfall areas. [18]

Temperatures that chill or freeze plant tissue are one of the three most important abiotic stresses that cause flower sterility and pod abortion. [19] Heat stress and

17 G Gibson G (2016) Innovative management techniques to reduce waterlogging. GRDC Update Papers. GRDC, https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/07/Innovative-management-techniques-to-reduce-waterlogging

18 V Devasirvatham, DK Tan, PM Gaur, RM Trethowan (2014) Chickpea and temperature stress: An overview. Legumes under Environmental Stress: Yield, Improvement and Adaptations 81.

19 A Maqbool, S Shafiq, L Lake (2010) Radiant frost tolerance in pulse crops—a review. Euphytica 172 (1), 1–12.

drought are also major factors. Chilling temperatures occur from 15°C to –1.5°C, and freezing temperatures are –1.5°C and below.

14.3.1 Impact of freezing range (less than –1.5°C)

Plants sensitive to freezing are damaged or killed by temperatures below –1.5°C. Damage from freezing commonly occurs due to ice forming into lattices within the intercellular spaces. The rigid ice structure extends as the temperature drops, and may penetrate cellular walls and membranes to an extent that cells are unable to repair themselves and the plant dies.

The tolerance of a plant to freezing varies greatly between different tissues, i.e. upper and lower leaves of the plant canopy, stems, meristems, and roots. Tolerance to freezing-range temperatures has been shown to decrease as the plant progresses from the seedling stage to flowering.

Freezing stress predominantly occurs during the seedling and early vegetative stages of crop growth.

Prolonged periods of freezing-range temperatures can prevent germination, reduce the vigour and vegetative biomass of the developing plant, and can be fatal to plants, especially those at the late vegetative and reproductive phenological growth stages.

The main effects of freezing temperatures on the developing seedling are related to membrane injury, which reduces respiration and photosynthesis, and causes loss of turgor, resulting in wilting and temperature-induced drought stress. Some observations have indicated that freezing can reduce seed size and cause seed-coat discolouration, probably due to stress conditions affecting the mobilisation of plant resources. [20]

14.3.2 Impact of chilling range (–1.5°C to 15°C)

In chickpeas, the upper limits of the chilling range are quite acceptable, and even optimum for early growth in some genotypes, but reproductive processes can become susceptible to damage from temperatures of around 15°C and lower.

In the Mediterranean-type environment of southern Australia, chickpea yields are limited by chilling-range temperatures during flowering, when chilling causes extensive flower and pod abortion. This is especially a problem for early-sown crops when growers are aiming for high yield potential (high biomass) and for early-flowering genotypes. The abortion of flowers and pods in late winter and early spring leads to low harvest index.

Desi chickpea seed can germinate in soil as cold as 5°C, but seedling vigour is greater if the soil temperature is at least 7°C. Kabuli chickpea seed is more sensitive to cold soils and should not be seeded into excessively wet soil or into soil with a temperature below 12°C at the placement depth.

In chickpeas, sensitivity to freezing- and chilling-range temperatures increases as the plant progresses from germination to flowering. Temperatures within the chilling range can limit the growth and vigour of chickpeas at all stages, but are considered most damaging to yield at the reproductive stages. Southern Australia is most affected by chilling-range temperatures at flowering. A prolonged period of chilling-range temperatures at any phenological stage of development in chickpeas has detrimental effects on the final seed yield.

During germination, chilling-range temperatures result in poor crop establishment, increased susceptibility to soil-borne pathogens, and reduced seedling vigour. At the seedling stage, long periods of chilling-range temperatures can retard the growth of the plant and, in severe cases, cause plant death. Visual symptoms of chilling injury at the seedling stage include the inhibition of seedling growth, accumulation of anthocyanin (red, purple and blue) pigments, waterlogged appearance with browning

20 JS Croser,, HJ Clarke,, KHM Siddique, TN Khan (2003). Low-temperature stress: implications for chickpea (*Cicer arietinum* L.) improvement. Critical Reviews in Plant Sciences 22 (2), 185–219, https://www.researchgate.net/publication/234520461_Low-Temperature_Stress_Implications_for_Chickpea_Cicer_arietinum_L_Improvement

of the mesocotyls, and the browning and desiccation of coleoptiles and undeveloped leaves. At the vegetative stage, chilling-range temperatures markedly slow down plant growth and dry matter production. Less dry matter production reduces the reproductive sink that the plant can support, which, in turn, reduces yield. Flower, pod, or seed abortion are further symptoms of chilling-range temperatures (Figure 2).

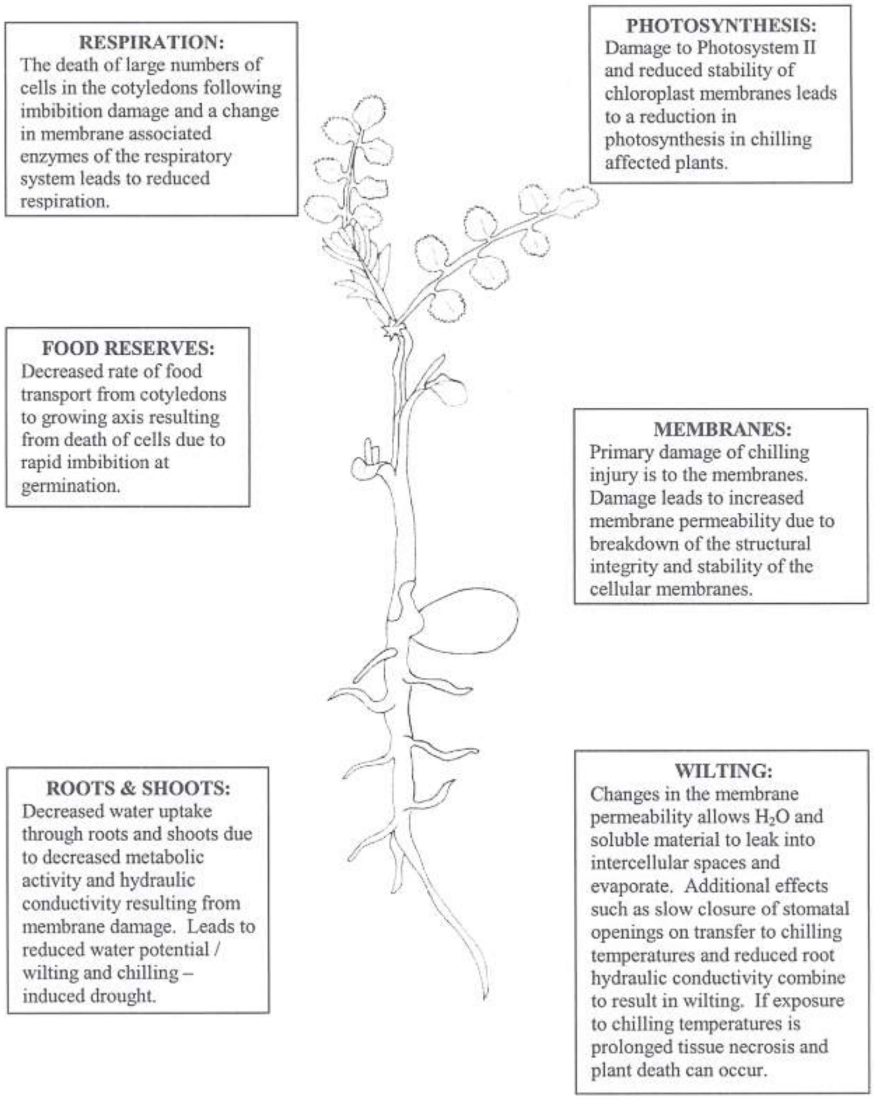


Figure 2: *Effect of chilling injury on chickpeas at the seedling to vegetative phenological stages.*

Source: Croser *et al.* 2003

Chilling range temperatures at the mid to late vegetative stage retard growth rate and reduce plant vigour. These effects are due to the same mechanisms that affect post-emergent seedling growth, that is, reduced respiration and photosynthesis, and in severe cases a loss of turgor and subsequent water stress.

Air temperature and photoperiod have a major influence on the timing of reproductive events in chickpea (Tables 6 and 7), with the rate of progress to flowering being a linear function of mean temperature. Pollen germination and vigour is affected by chilling-range temperatures. [21]

21 JS Croser, ,HJ Clarke, KHM Siddique, TN Khan (2003) Low-temperature stress: implications for chickpea (*Cicer arietinum* L.) improvement. Critical Reviews in Plant Sciences 22 (2), 185–219, https://www.researchgate.net/publication/234520461_Low-Temperature_Stress_Implications_for_Chickpea_Cicer_arietinum_L_Improvement

Table 6: *Effect of chilling-range temperatures on chickpea reproduction.*

Effect	Description
Flower shedding/floral abortion Pod shedding/drop	Sudden low temperatures (0° to 10°C) during flowering induces flower shedding, which causes partitioning of assimilates to vegetative growth, resulting in lowered Harvest Index. Major cause of low pod and seed set in subtropical South Asia and Australia.
Interrupted pollen tube growth	Temperatures up to 25°C shown to interrupt pollen tube growth. Failure of fertilisation results from poor germination and slow growth of pollen tubes in susceptible genotypes at low temperatures (Figure 4).
Lowered pollen viability	Pollen in tolerant genotypes more viable (90%) compared to susceptible genotypes (60%). Two stages of pollen sensitivity at 5 and 9 days before anthesis have been identified.
Reduced ovule size	Ovules in flowers opening on cool days were 9–45% smaller than warm day ovules – more pronounced in chilling susceptible than tolerant genotypes.
Reduced pistil size	Heterostyly – the distance between the anther and stigma at the time of flower opening is greater in sensitive than in tolerant genotypes.
Reduced stigmatic esterase activity	Reduced esterase activity was identified in susceptible genotypes suggesting the stigmas were less receptive to pollen tube growth.
Delayed anther dehiscence	Anther dehiscence is delayed by chilling temperatures, reducing fertilisation events.
Reduced pollen germinability	Possibly due to smaller amount of storage material in pollen from sensitive genotypes.
Reduced pollen turgor	Turgidity is an absolute requirement for germination. Pollen cells with leaking membranes cannot become turgid and germinate.

Source: Croser *et al.* 2003

Table 7: *Effect of chilling range temperatures at flowering on chickpea productivity in Mediterranean climate.*

Date of planting	Date of 50% flowering	Mean daily temperature (°C) at 50% flowering	Number of aborted flowers (m-2)	Biological yield (t/ha-1)	Seed yield (t/ha-1)	Harvest index
17 May	19 August	12.5	800	6.76	1.25	0.18
31 May	1 September	13.6	500	5.34	1.13	0.21
14 June	14 September	14.7	200	4.84	1.12	0.23
30 June	29 September	16.8	0	3.98	1.11	0.28
20 July	6 October	17.7	0	3.23	0.94	0.29

Source: Modified from Siddique, Marshall and Sedgley (1983) [22]

Exposure to prolonged periods of temperatures at the lower end of the chilling range can cause poor germination, slow growth, flower shedding, and pod abortion, and in severe cases cell necrosis and plant death. The resulting yield penalty or reduction in harvest index varies dramatically in the field, but can be substantial (Photo 7).

Photo 7: *Chilling damage in chickpea- sown paddock.*

Source: Pulse Breeding Australia

In Australia, flower shed and pod abortion due to chilling-range temperatures at flowering is a major cause of poor yield. It should be noted that this is in combination with terminal drought. Early sowing (winter) is essential in these environments in order to maximise yield and avoid terminal soil-moisture stress.

The development of cultivars with a higher degree of cold tolerance would facilitate the spread of chickpea-growing regions to both higher altitudes and colder latitudes and therefore are worthy of considerable agronomic and breeding attention. [23]

Tolerance to low temperature

Low temperature at flowering is a major constraint to improving yields of chickpeas in many regions of the world. In particular, cool dryland environments such as that of southern Australia would benefit from cultivars with the ability to flower and set pods early in the growing season before soil moisture becomes a limiting factor.

22 KHM Siddique, C Marshall, RH Sedgley (1983) Temperature and leaf appearance in chickpea. International Chickpea Newsletter 8, 14–15.

23 JS Croser, ,HJ Clarke, KHM Siddique, TN Khan (2003) Low-temperature stress: implications for chickpea (*Cicer arietinum* L.) improvement. Critical Reviews in Plant Sciences 22 (2), 185–219.

Desi types generally suffer less damage from low temperatures at germination than kabuli types. Research overseas and within Australia has demonstrated a range of cold tolerance among chickpea varieties. In parts of the world where chickpeas are grown as a spring crop because of the very cold winter, varieties have been developed that tolerate freezing conditions during vegetative growth. These varieties can be sown in autumn, survive over winter, and are ready to flower and set pods when temperatures rise in summer.

However, chickpea varieties resistant to low temperatures during flowering have not yet been developed. Some genotypes from India are less sensitive than those currently grown in Australia, and these are being utilised in chickpea-breeding programs.

Controlled-environment studies at the University of Western Australia have identified two stages of sensitivity to low temperatures in chickpeas. The first occurs during pollen development in the flower bud, and results in infertile pollen even in open flowers. The second stage occurs at pollination, when pollen sticks to the female style, and produces a tube that grows from the pollen down the style to the egg (Photo 8).

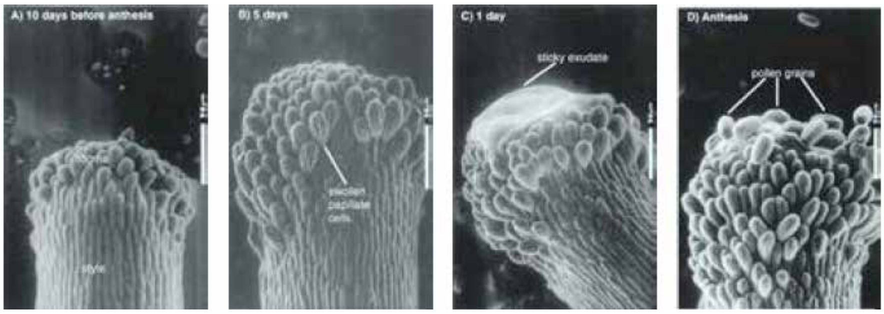


Photo 8: *Development of the style and stigma of chickpea flowers taken with an electron microscope.*

Photo H. Clarke, UWA

At low temperatures pollen tubes grow slowly, fertilisation is less likely and the flower often aborts. The rate of pollen-tube growth at low temperature is closely related to the cold tolerance of the whole plant. This trait can therefore be used to select more tolerant varieties (Figure 3).

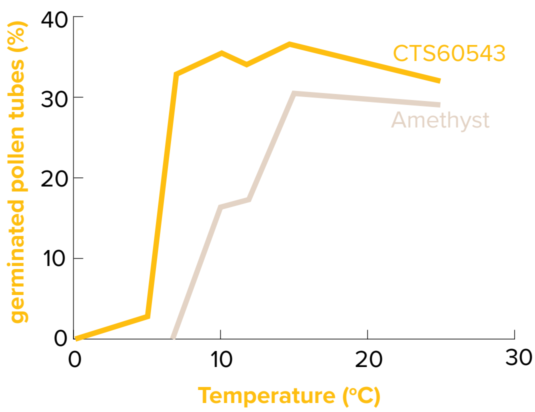


Figure 3: *Proportion of pollen germination at various temperatures in cold-sensitive (Amethyst) and cold-tolerant (CTS60543) varieties.*

The critical average daily temperature for abortion of flowers in most varieties currently grown in Australia is about 15°C. New hybrids that set pods at ~13°C are being developed.

In the field, cold-tolerant varieties set pods about 1–2 weeks earlier than most current varieties. As well as conventional methods for plant improvement, DNA-based techniques are also being investigated. [24]

IN FOCUS

Response of chickpea genotypes to low-temperature stress during reproductive development

Clarke and Siddique (2004) showed temperatures of less than 15°C affect both the development and the function of reproductive structures in the chickpea flower.

The function of pollen derived from chilling-sensitive plants is the most affected by low-temperature stress, and particularly the growth of the pollen tubes down the style before fertilisation occurs. Pollen tubes derived from chilling-tolerant plants continue to grow down the style under low-temperature stress.

Two periods of sensitivity to low temperature were identified during pollen development (sporogenesis) in both Amethyst (cold tolerant) and CTS60543 (cold sensitive), each of which resulted in chilling-stressed plants having flowers with only 50% viable pollen (Figure 4 top).

The researchers estimated that the first stage of sensitivity occurred nine days before anthesis. The significant decrease in pollen viability coincided with lower podset (approximately 70%) in both genotypes. This was followed by a recovery in pollen viability and podset.

The second period of sensitivity occurred 4–6 days before anthesis. In Amethyst a dramatic drop to 40% podset correlated with the decrease in pollen viability (Figure 4 bottom), while normal pods formed at other nodes before and after this chilling-sensitive stage. Despite lower pollen viability in CTS60543, podset was not affected at this time, and all of the flowers gave rise to full pods in this genotype.

24 Clarke, H. J., & Siddique, K. H. M. (2004). Response of chickpea genotypes to low temperature stress during reproductive development. Field Crops Research, 90(2), 323-334

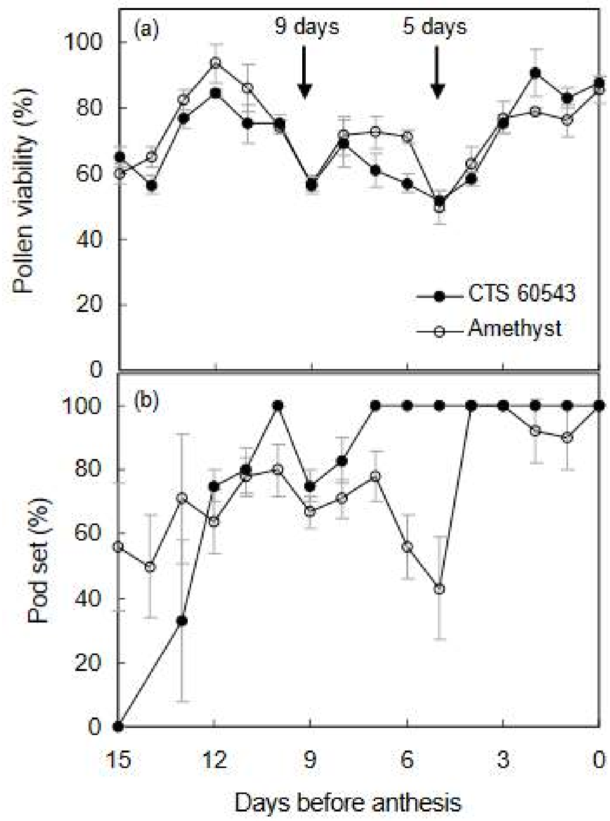


Figure 4: *The effects of stressing plants with a temperature of 3°C during flower development in tolerant and sensitive genotypes. (A) Pollen viability. (B) Podset. Arrows indicate susceptible periods at 9 days and 5–6 days before anthesis.*

Temperatures from 7–25°C did not affect the proportion of pollen grains which germinated after four-hour incubation in vitro, and 80–90% germination occurred in all of the genotypes examined (Figure 5). The percentage that germinated was significantly lower at 3°C than at other temperatures, but no significant difference was measured between genotypes at this temperature when samples were examined at four hours.

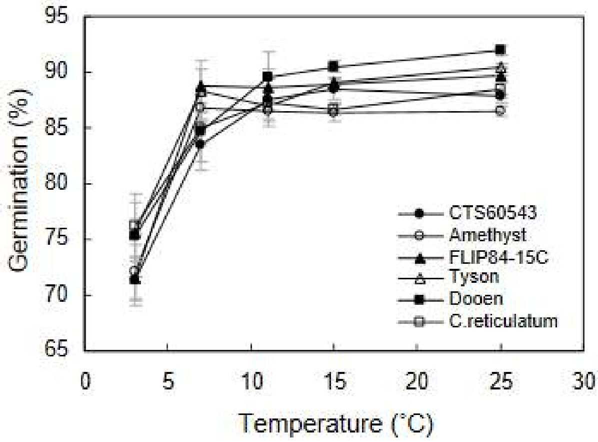


Figure 5: *The effect of temperature on pollen germination in chickpea genotypes after four-hour culture in lab conditions.*

A second experiment was therefore designed to examine the rate of germination with selected genotypes at 3°C in the period up to four hours *in vitro*. The researchers found that low temperature delayed the onset of germination by 20–40 minutes compared to the control at 25°C, and it decreased the rate at which the pollen germinated (Figure 6). No significant difference was measured in germination between genotypes in the first 40 minutes in culture. After this time the percentage that germinated was significantly different between genotypes.

However, no link was observed between the rate of pollen germination in vitro at 3°C and the chilling tolerance of the whole plant. In fact, pollen from CTS60543 was slightly slower to begin germination.

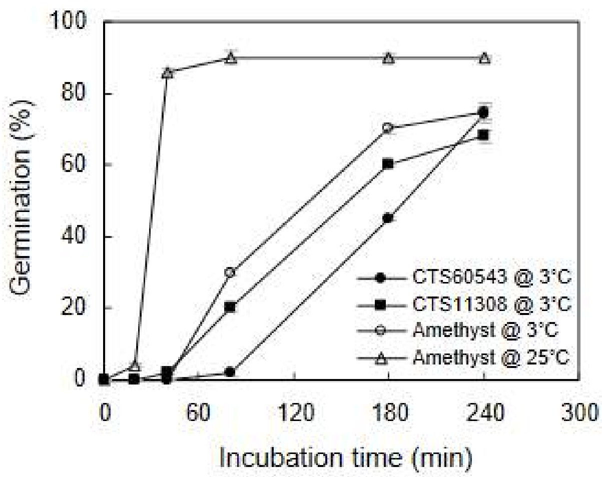


Figure 6: *Rate of pollen germination during 4 hour culture in lab conditions at low temperature (3°C) in sensitive and tolerant genotypes.*

Figure 7 illustrates a greater number of pollen tubes in the style in CTS60543 compared to Amethyst when hand-pollinated plants are stressed at 7°C for 24 h after pollination. [25]

25 HJ Clarke, KHM Siddique (2004) Response of chickpea genotypes to low temperature stress during reproductive development. Field Crops Research 90 (2), 323–334.

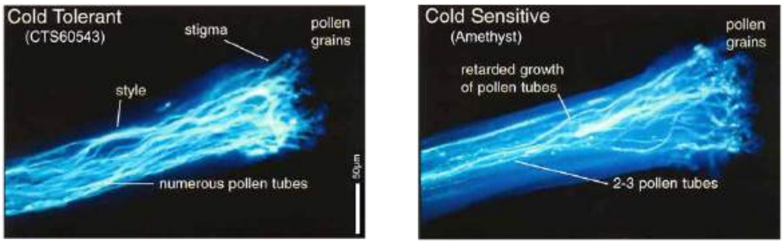


Figure 7: *More pollen tubes of CTS60543 grow down the style to the ovary at low temperature stress (7°C) compared to Amethyst. Styles were fixed 24 h after pollination and stained with aniline blue.* [26]

14.3.3 Heat stress

High-temperature stress in chickpeas causes substantial loss (Photo 9) in crop yield due to damage to the reproductive organs, increased rate of plant development, and reduced length of the reproductive period. [27]

Photo 9: *Chickpea varieties planted under hot conditions: heat-sensitive plant with no pods (left) and heat-tolerant plant with healthy pods (right).*

Photo: D. Tan, University of Sydney

Chickpeas are sensitive to heat stress (36/16°C or higher as day/night temperatures) in their reproductive stage with potential loss of yields at high temperatures. The anthers of heat-sensitive genotypes have been found to have reduced synthesis of sugars due to the inhibition of the appropriate enzymes. Consequently, affected plant pollen can have considerably lower sucrose levels, which results in reduced pollen function, impaired fertilisation and poor podset in the heat-sensitive genotypes.

IN FOCUS

Reproductive failures in heat-stressed chickpeas are associated with impaired sucrose metabolism in leaves and anthers

In the heat-stressed plants, phenology accelerated as days to flowering and podding, and biomass decreased significantly. The significant reduction in podset was associated with reduced pollen viability, pollen

26 HJ Clarke, KHM Siddique (2004) Response of chickpea genotypes to low temperature stress during reproductive development. Field Crops Research 90 (2), 323–334.

27 Y Gan, J Wang, SV Angadi, CL McDonald (2004) Response of chickpea to short periods of high temperature and water stress at different developmental stages. Proceedings of the 4th International Crop Science Congress, Brisbane. http://www.cropscience.org.au/icsc2004/poster/1/3/3/603_ganyd.htm

load, pollen germination (*in vivo* and in *vitro*) and stigma receptivity in all four genotypes researched.

Heat stress inhibited pollen function more in the sensitive genotypes than in the tolerant ones, and consequently showed significantly less podset.

Heat stress significantly reduced stomatal conductance, leaf water content, chlorophyll, membrane integrity and photochemical efficiency, with a larger effect on heat-sensitive genotypes. Because the plants were heat stressed, rubisco (a carbon-fixing enzyme) along with sucrose phosphate synthase (SPS) and sucrose synthase (SS) (sucrose-synthesising enzymes) decreased significantly in the leaves, and this led to reduced sucrose content. Invertase, a sucrose-cleaving enzyme, was also inhibited along with SPS and SS. The inhibition of these enzymes was significantly greater in the heat-sensitive genotypes. Concurrently, the anthers of these genotypes had significantly less SPS and SS activity and thus, less sucrose content. Pollen had considerably lower sucrose levels, resulting in reduced pollen function, impaired fertilisation and poor podset. [28]

Chickpea pollen grains are more sensitive to heat stress than the stigma. High temperatures have been found to reduce pollen production in each flower, the amount of pollen germination, podset and seed number and size (Photo 10). [29]

Photo 10: *Comparison of seed size under heat stress. The larger seeds (left) are from non-stressed plants, and the smaller seeds (right) from heat-stressed conditions.*

Photo: V. Devasirvatham 2015

Heat stress during the reproductive stage can cause significant yield loss. Very little of the germ plasm will set pods when temperatures exceed 36°C. During the reproductive stage heat stress can cause asynchrony of male and female floral organ development and impairment of male and female floral organs resulting in lower yields. [30]

28 N Kaushal, R Awasthi, K Gupta, P Gaur, KH Siddique, H Nayyar (2013) Heat-stress-induced reproductive failures in chickpea (*Cicer arietinum*) are associated with impaired sucrose metabolism in leaves and anthers. Functional Plant Biology, 40 (12), 1334–1349, https://www.researchgate.net/publication/265209338_Heat-stress-induced_reproductive_failures_in_chickpea_Cicer_arietinum_are_associated_with_impaired_sucrose_metabolism_in_leaves_and_anthers

29 V Devasirvatham, PM Gaur, N Mallikarjuna, RN Tokachichu, RM Trethowan, DK Tan (2012) Effect of high temperature on the reproductive development of chickpea genotypes under controlled environments. Functional Plant Biology, 39 (12), 1009–1018, http://www.publish.csiro.au/FP/pdf/FP12033

30 V Devasirvatham, DKY Tan, PM Gaur, TN Raju, RM Trethowan (2012) High temperature tolerance in chickpea and its implications for plant improvement. Crop and Pasture Science, 63 (5), 419–428, http://www.plantstress.com/Articles/up_heat_files/Heat tolerance (chickpea) 2012.pdf

14.3.4 Heat and water stress

IN FOCUS

Response of Chickpea to Short Periods of High Temperature and Water Stress at Different Developmental Stages.

This study was conducted to determine the effect of short periods of high temperature and water stress on pod production, seedset and yield of chickpeas.

Plants stressed at 35/16°C during flowering produced 53% fewer fertile pods on the main stem and 22% fewer pods on the branches than those kept at 20/16°C. Nearly 90% of the pods formed during stress were infertile. Due to high temperature stress, kabuli crop filled 58% of the pods formed and decreased seeds pod-1 by 26% from the check. Consequently, desi chickpea seed yield decreased by 54% when stressed during pod development and 33% when stressed during flowering. Kabuli chickpea seed yield decreased by 50% when stressed during pod formation and 44% when stressed during flowering. [31]

14.4 Drought stress

Chickpeas are an important winter pulse crop for the neutral-to-alkaline heavy-textured soils in both the Mediterranean climatic region and the summer-rainfall region of Australia. Mediterranean chickpea-growing regions generally have cool, wet winters, and in spring, increasing temperatures and reduced rainfall result in a terminal drought for most crops. Unlike many other crops, chickpeas are unable to escape terminal drought through rapid development because low temperatures (<15°C) often cause flower and pod abortion, especially in cool southern areas. [32]

Yield losses can be the result of intermittent drought during the vegetative phase, due to drought during reproductive development, or due to terminal drought at the end of the crop cycle. [33]

After disease, the major constraint to greater chickpea production is its sensitivity to the end-of-season drought that occurs in both the Mediterranean climates and when grown on stored soil moisture in the summer-rainfall region of Australia. Terminal drought in a Mediterranean climate affects chickpeas because rainfall starts decreasing and evaporation starts increasing just as they enter their reproductive stage. The summer-rainfall regions are more dependent on stored soil moisture, and terminal drought occurs because the soil moisture is exhausted during the seed-filling stage.

Terminal drought reduces leaf photosynthesis in chickpeas before seed growth commences, so that seed filling is, in part, dependent on carbon and nitrogen accumulated prior to podding. For example, under terminal drought, more

31 Y Gan, J Wang, SV Angadi, CL McDonald (2004) Response of chickpea to short periods of high temperature and water stress at different developmental stages. Proceedings of the 4th International Crop Science Congress, Brisbane. http://www.cropscience.org.au/icsc2004/poster/1/3/3/603_ganyd.htm

32 RJ Jettner, SP Loss, KHM Siddique, RJ French (1999) Optimum plant density of desi chickpea (*Cicer arietinum* L.) increases with increasing yield potential in south-western Australia. Crop and Pasture Science, 50 (6), 1017–1026, https://www.researchgate.net/publication/234520394_Optimum_plant_density_of_desi_chickpea_Cicer_arietinum_L_increases_with_increasing_yield_potential_in_south-western_Australia

33 A Mafakheri, A Siosemardeh, B Bahramnejad, PC Struik, Y Sohrabi (2010) Effect of drought stress on yield, proline and chlorophyll contents in three chickpea cultivars. Australian Journal of Crop Science, 4 (8), 580–585, http://eprints.icrisat.ac.in/422/1/AustralianJofCropScience.pdf

than 90% of the seed nitrogen in chickpea comes from pre-podding sources, particularly leaves. [34]

IN FOCUS

How chickpeas respond to terminal drought: leaf stomatal conductance, pod abscisic acid concentration, and seedset

At flowering, pod production and seedset, chickpeas are sensitive to drought stress. It is already known that drought stress impairs pollen viability and the function of the stigma and style, but it was not clear whether flower and pod abortion was due to the failure of the pollen tube to reach the ovary, or to other factors.

In one study, pollen viability and germination decreased when the fraction of transpirable soil water (FTSW) decreased below 0.18 (i.e. 82% of the plant-available soil water had been transpired); however, at least one pollen tube in each flower reached the ovary. The young pods which developed from flowers produced when the FTSW was 0.50 had viable embryos, but contained higher concentrations of abscisic acid (ABA) than those of the well-watered plants; all pods ultimately aborted in the drought treatment. Cessation of seedset at the same soil-water content at which stomata began to close and ABA increased strongly suggested a role for ABA signalling in the failure to set seed either directly through abscission of developing pods or seeds or indirectly through the reduction of photosynthesis and assimilate supply to the seeds. [35]

Drought after podding is a common feature of chickpea production in southern Australia. One study investigated the effect of water stress, which was imposed after podding, on yield and on the accumulation of amino acids and soluble sugars in seeds. Although terminal water stress decreased the total plant dry mass by 23% and seed yield by 30%, it had no effect on the mass of individual pods and seeds which remained on the plant after the imposition of stress treatment. The deleterious effect of water stress on yield was due to increased pod abortion and a decrease in pod formation. Water stress improved the seed's nutritive value in terms of higher accumulation of soluble sugars, amino acids and proteins. [36]

Plants grown under drought conditions have a lower stomatal conductance in order to conserve water. (Stomatal conductance is the rate at which carbon dioxide enters and water vapour exits the stomata of a leaf.) Consequently, CO_2 fixation is reduced and photosynthetic rate decreases, resulting in less assimilate production for growth and yield of plants. Drought stress during vegetative growth or anthesis significantly decreases chlorophyll and therefore photosynthesis. Drought stress at anthesis phase can reduce seed yield more severely than when it occurs during the vegetative stage. [37]

34 JA Palta, AS Nandwal, S Kumari, NC Turner (2005) Foliar nitrogen applications increase the seed yield and protein content in chickpea (*Cicer arietinum* L.) subject to terminal drought. Crop and Pasture Science, 56 (2), 105–112, http://www.publish.csiro.au/AR/pdf/AR04118

35 J Pang, NC Turner, T Khan, YL Du, JL Xiong, TD Colmer, R Devilla, K Stefanova, KHM Siddique (2016) Response of chickpea (*Cicer arietinum* L.) to terminal drought: leaf stomatal conductance, pod abscisic acid concentration, and seed set. Journal of Experimental Botany. Published online 19 April 2016. DOI 10.1093/jxb/erw153, http://jxb.oxfordjournals.org/content/early/2016/04/19/jxb.erw153.full

36 MH Behboudian, Q Ma, NC Turner, JA Palta (2001) Reactions of chickpea to water stress: yield and seed composition. Journal of the Science of Food and Agriculture, 81 (13), 1288–1291, http://onlinelibrary.wiley.com/doi/10.1002/jsfa.939/abstract

37 A Mafakheri, A Siosemardeh, B Bahramnejad, PC Struik, Y Sohrabi (2010) Effect of drought stress on yield, proline and chlorophyll contents in three chickpea cultivars. Australian Journal of Crop Science, 4 (8), 580–585, http://eprints.icrisat.ac.in/422/1/AustralianJofCropScience.pdf

IN FOCUS

Variation in pod production and abortion among chickpea cultivars under terminal drought

The researchers studied the effect of terminal drought on the dry matter production, seed yield and its components, including pod production and pod abortion, was investigated in chickpeas. Two desi chickpea cultivars (with small, angular and dark brown seeds) and two kabuli cultivars (with large, rounded and light coloured seeds) differing in seed size were grown in a greenhouse where the temperature was controlled and water stress was applied by withholding irrigation one week after podset commenced (early podding water stress, ES), two weeks after (mid-podding water stress, MS) or three weeks after (late-podding water stress, LS). In addition, the pod and seed growth of well-watered plants was followed for the first 19 days after podset.

The results show that pod abortion is one of the main reasons for decreases in seed yield in chickpeas exposed to terminal drought and that, irrespective of differences in phenology, kabuli types have greater pod abortion than desi types when water deficits develop shortly after first podset. [38]

A lack of water can also impair light interception and light-use efficiency (and photosynthesis) by affecting chickpea development during leaf expansion. Therefore, the timing of water stress during chickpea canopy development will determine how well the plant intercepts and uses light. [39]

IN FOCUS

Physiological responses of chickpea genotypes to terminal drought in a Mediterranean-type environment

Two field experiments were carried out to investigate the effects of terminal drought on chickpeas grown under water-limited conditions in the Mediterranean-climatic region of Western Australia.

In the first experiment, five desi chickpeas (with small, angular seeds) and one kabuli chickpea (with large, round seeds) were grown in the field with and without irrigation after flowering. In the second experiment, two desi and two kabuli cultivars were grown in the field with either irrigation or under a rainout shelter during pod filling. In bother experiments, researchers measured leaf-water potential, dry-matter partitioning after podset and yield components, and in the first experiment they also measured growth before podset, photosynthesis, pod-water potential and leaf osmotic adjustment.

In the rain-fed plants, leaf-water potential decreased below −3 MPa while photosynthesis decreased to about a tenth of its maximum at the start of seed filling. Osmotic adjustment varied significantly among genotypes.

38 L Leport, NC Turner, SL Davies, KHM Siddique (2006) Variation in pod production and abortion among chickpea cultivars under terminal drought. European Journal of Agronomy, 24 (3), 236–246, http://www.sciencedirect.com/science/article/pii/S1161030105000985

39 s Thoma, GL Hammer (1995) Growth and yield response of barley and chickpea to water stress under three environments in southeast Queensland. II. Root growth and soil water extraction pattern. Aust J of Ag Research. 46 (1), 35–48.

Although flowering commenced from about 100 days after sowing (DAS) in both experiments, podset was delayed until 130–135 DAS in the first experiment, but started at 107 DAS in the second experiment. The shortage of water reduced seed yield by 50–80%, due to a reduction in seed number and seed size.

Apparent redistribution of stem and leaf dry matter during pod filling varied from 0–60% among genotypes, and suggests that this characteristic may be important for a high harvest index and seed yield in chickpea. [40]

14.4.1 Managing for drought

Long-term historical records indicate that our climate is becoming progressively warmer and dryer. This trend is expected to continue due to increased levels of greenhouse gas in the atmosphere, with dry seasons likely to become more frequent over southern Australia. [41]

For crops exposed to terminal drought, the application of nitrogen fertiliser to the soil during podset and seed filling is unlikely to assist in delaying the withdrawal of nitrogen from the leaves and maintaining leaf photosynthesis because nitrogen is not taken up from dry soil. However, foliar applications of urea have been effective in increasing the nitrogen availability for seed filling. [42]

IN FOCUS

Foliar nitrogen increases the seed yield and protein content in chickpeas subject to terminal drought

Researchers hypothesised that applying urea to the leaves of chickpeas may increase the amount of nitrogen available for seed filling when chickpeas are grown under terminal drought. The study was conducted in a glasshouse where they were able to mimic terminal drought.

Nitrogen was applied at: (i) first flower; (ii) 50% flowering; (iii) 50% pod set; and (iv) the end of podding; with (v) being an unsprayed control. Terminal drought was induced from pod set onward.

Foliar applications of urea at first flower and at 50% flowering, before terminal drought was induced, resulted in greater yield and higher seed protein content.

The increase in yield occurred because of an increase in the number of pods with more than one seed (rather than from greater numbers of pods per plant or increased seed size). This indicated that more seeds survived terminal drought. The increase in the seed-protein content occurred because there was more nitrogen available for seed filling.

The foliar application of urea during flowering, before terminal drought was induced, resulted in 20% more biomass at maturity. This suggested that growth that occurred before the water shortage gave the plant more carbon resources which allowed it to sustain seed filling even under conditions of terminal drought.

40 L Leport, NC Turner, RJ French, MD Barr, R Duda, SL Davies, D Tennant, KHM Siddique (1999) Physiological responses of chickpea genotypes to terminal drought in a Mediterranean-type environment. European Journal of Agronomy, 11 (3), 279–291, http://www.sciencedirect.com/science/article/pii/S1161030199000398

41 DAFWA. Drought, https://www.agric.wa.gov.au/climate-land-water/climate-weather/drought

42 JA Palta, AS Nandwal, S Kumari, NC Turner (2005) Foliar nitrogen applications increase the seed yield and protein content in chickpea (*Cicer arietinum* L.) subject to terminal drought. Aust J Ag Research. 56 (2), 105–112.

Foliar applications of urea at 50% podset and at the end of podding did not affect the yield or seed-protein content, primarily because the uptake of nitrogen was limited by the leaf senescence that occurs with the development of terminal drought.

The results indicate the potential to increase yields of chickpeas by applying foliar nitrogen near flowering in environments in which terminal droughts reduce yield. [43]

14.4.2 Adaptation to drought stress

There are three strategies of crop adaptation to drought-stressed environments, all of which are useful in the Mediterranean climate:

1. Drought escape, where the crop completes its life cycle before the onset of terminal drought.
2. Drought avoidance, where the crop maximises its water uptake and minimises water loss.
3. Drought tolerance, where the crop continues to grow and produce even with reduced water. [44]

Despite the ability of chickpeas to thrive in a wide range of environments, there has been little development of specifically adapted varieties, and at this stage the same cultivar may be seen in farmers' fields from Queensland to Western Australia. The basis of the wide adaptation in chickpeas is important as new cultivars are developed.

Chickpeas may adapt to drought stress by maximising water uptake through continuous root growth up to seed filling, and by maintaining substantial water uptake until the fraction of extractable moisture in the root profile falls to 0.4. [45] In addition to these strategies, the early sowing of chickpeas in south-western Australia helps to develop a large green area that rapidly covers the ground. This means that plants absorb a significant proportion of photosynthetic-active radiation early in the season when vapour-pressure deficits are low, and that they use more water in the post-flowering period. [46] Consequently, such crops produce large biomass and grain yield. Application of supplemental irrigation at flowering and early pod-filling can relieve drought stress and substantially increase seed yield. [47]

Plants can partly protect themselves against mild drought stress by accumulating osmolytes (compounds that affect osmosis). Proline is one of the most common compatible osmolytes in drought-stressed plants. It does not interfere with normal biochemical reactions, and allows the plants to survive under stress. The more drought-tolerant varieties have a greater capacity to accumulate proline, which gives them a buffer against the effects of drought. [48]

One study found that a strain of symbiotic rhizobia (nitrogen-fixing bacteria) was effective in root-nodule symbiosis, partially alleviated decreased growth and yield, and increasing root biomass of chickpeas under drought stress. [49]

43 JA Palta, AS Nandwal, S Kumari, NC Turner (2005) Foliar nitrogen applications increase the seed yield and protein content in chickpea (*Cicer arietinum* L.) subject to terminal drought. Aust J Ag Research, 56 (2), 105–112.

44 MM Ludlow (1989) Strategies of response to water stress. Structural and Functional Responses to Environmental Stresses, 269–281.

45 KHM Siddique , RH Sedgley (1987) Canopy development modifies the water economy of chickpea (*Cicer arietinum* L.) in south-western Australia. Aust J Ag Research, 37 (6), 599–610.

46 Cited in H Zhang, M Pala, T Oweis, H Harris (2000) Water use and Water Use Efficiency of chickpea and lentil in a Mediterranean environment. Crop and Pasture Science, 51 (2), 295–304.

47 H Zhang, M Pala, T Oweis, H Harris (2000) Water use and Water Use Efficiency of chickpea and lentil in a Mediterranean environment. Crop and Pasture Science, 51 (2), 295–304.

48 A Mafakheri, A Siosemardeh, B Bahramnejad, PC Struik, Y Sohrabi (2010) Effect of drought stress on yield, proline and chlorophyll contents in three chickpea cultivars. Australian Journal of Crop Science, 4 (8), 580–585, http://eprints.icrisat.ac.in/422/1/AustralianJofCropScience.pdf

49 A Bano, R Batool,, F Dazzo. (2010) Adaptation of chickpea to desiccation stress is enhanced by symbiotic rhizobia. Symbiosis, 50 (3), 129–133, http://link.springer.com/article/10.1007/s13199-010-0051-9/fulltext.html

IN FOCUS

The role of phenology in adaptation of chickpeas to drought

Chickpeas are grown from autumn to early summer in both Mediterranean-type climates with winter dominant rainfall, and in sub-tropical climates with stored soil moisture and summer-dominant rainfall. In both environments, water shortages can occur at any time during the growing season, but terminal drought predominates. A study of 73 genotypes conducted over two years showed that high-yielding genotypes flowered early, podded early and had a relatively long flowering period at most, thought not all, low-yielding sites. Thus drought escape was an important phenological characteristic for successful chickpea production at sites with terminal drought. However, these characteristics did not predominate at a site where drought was severe throughout the growing period.

Studies under rain-fed conditions at a dry site in Western Australia have shown that a high degree of biomass redistribution from leaves to stems to the pod is associated with high yield. This suggests that, in addition to rapid phenological development, physiological mechanisms play a role in the adaptation of chickpeas to water-limited environments. [50]

Breeding chickpeas for drought tolerance and disease resistance

A GRDC-funded project, Breeding chickpea for drought tolerance and disease (project ICA00008), aimed to enhance production, productivity and yield stability of chickpeas grown in Mediterranean and similar Australian environments by improving genetic factors and agronomic options. Most chickpea cultivars grown in Mediterranean-type environments are susceptible to Ascochyta blight, affected by terminal drought, and susceptible to cold at the vegetative and flowering stages.

One of the reasons the project was important is that the identification of new sources of resistance or tolerance to Ascochyta blight, drought and Fusarium wilt is a continuous process, as the pressure from these stresses is continuously evolving.

The researchers sought to validate the results of a previous project, and to provide more efficient and reliable screening tools for the evaluation of germ plasm and breeding materials.

They shared new materials and methodologies with National Agricultural Research Systems in west Asia and north Africa, and with pulse-breeding programs in Australia.

This project delivers improved germ plasm to make chickpeas better able to combat these stressors. It also delivered germ plasm for other desirable traits, such as making the plant better suited to mechanical harvesting. Resistance to Ascochyta blight, to drought, and to Fusarium wilt were bred into adapted Australian cultivars, and the newly derived progenies are being advanced at International Center for Agricultural Research in Dry Areas (ICARDA).

These materials are shared with Australian partners at the advanced stage, to screen under local conditions. Selected lines will be used either in crossing programs or for testing in yield trials for direct release as cultivars. The pathway to the market is through the release and adoption of improved chickpea varieties.

50 J Berger, NC Turner, RJ French (2003) The role of phenology in adaptation of chickpea to drought. Solution for a Better Environment. Proceedings of the 11th Australian Agronomy Conference, Geelong, Victoria, Australia. February 2003, 2–6, http://www.regional.org.au/au/asa/2003/c/4/turner.htm

All these efforts are contributing to widen the genetic base of Australian chickpeas, which will give the industry more plasticity in the face of future threats to production. Farmers who adopt the new varieties developed in these materials derived from this project will contribute to greater and more sustainable chickpea production, and thereby to raising the level of the chickpea industry in Australia. [51]

14.5 Other environmental issues

14.5.1 Salinity

One of the most significant causes of soil degradation in Australia is salinity, which is the presence of dissolved salts in soil or water. It occurs when the water table rises, bringing natural salts to the surface; in sufficient quantity, the salts become toxic to most plants. They cause iron toxicity in plants and impede their ability to absorb water (Photos 11 and 12). Salinity, a major abiotic stress, is a major environmental production constraint in many parts of the world. In Australia, saline soils have been caused by extensive land clearing, predominantly for agricultural purposes. Chickpeas are extremely sensitive to salinity, and can have difficulty accessing water and nutrients from saline layers in the soil. This effectively limits water extraction from the subsoil, and consequently limits yields. Salinity impairs vegetative growth in chickpeas, but reproductive growth is particularly salt sensitive.

Photo 11: *Salt effects seen on soil and in subsequent chickpea growth.*

Source: Pulse Breeding Australia

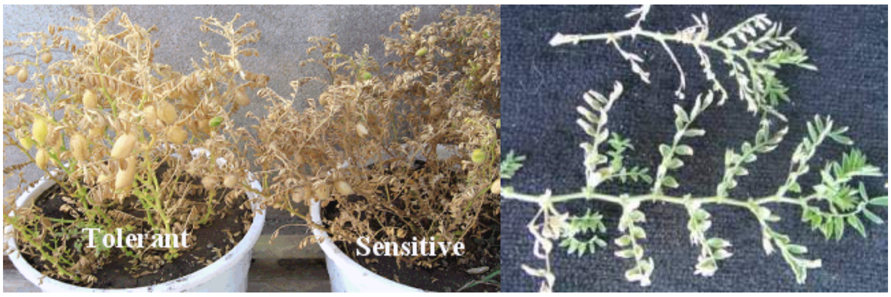


Photo 12: *Left: pod setting in saline soils in a salt-tolerant and a salt-sensitive genotype. While the biomass of the two genotypes may be similar, podset can be markedly different.* [52] *Right: typical salt effects on chickpea leaves.*

Photos: left - V Vadez *et al* (2006)52; right - Pulse Australia.

Nut grass (Photo 13) is often a good indicator of increased salt levels.

51 GRDC. Breeding chickpea for drought tolerance and disease resistance. Project ICA00008, http://projects.grdc.com.au/projects.php?action=view_project&project_id=1238

52 V Vadez, L Krishnamurthy, PM Gaur, HD Upadhyaya, DA Hoisington, RK Varshney, NC Turner, KHM Siddique (2006) Tapping the large genetic variability for salinity tolerance in chickpea. Proceeding of the Australian Society of Agronomy. Meeting 10–14 September 2006, http://agronomyaustraliaproceedings.org/images/sampledata/2006/concurrent/environment/4561_vadez.pdf

Photo 13: *Nut grass growing in saline soil.*

Photo: Pulse Australia

All current varieties of chickpeas are considered highly sensitive to salinity. Levels of electrical conductivity (EC) >1.5 dS/m will cause a yield reduction in chickpeas (Table 8). During growth chickpeas are very sensitive to salinity, with the most susceptible genotypes dying in just 25 mM of sodium chloride (NaCl), and resistant genotypes unlikely to survive 100 mm NaCl in hydroponics. They are more tolerant at germination, with some genotypes tolerating 320 mM NaCl. When chickpeas are grown in a saline medium, Cl–, which is secreted from glandular hairs on the leaves, stems and pods, is present in higher concentrations in the shoots than Na+. Salinity reduces the amount of water extractable from soil by a chickpea crop and induces osmotic adjustment, which is greater in the nodules than in the leaves or roots. Chickpea rhizobia show a higher salt resistance than chickpea plants, and salinity can cause large reductions in nodulation, nodule size and N_2-fixation capacity. [53]

Table 8: *Crop tolerances to salinity (EC: mmhos/cm = dS/m = mS/cm).*

Crop	Expected yield reduction (%)				
	0	**10**	**25**	**50**	**100**
Chickpeas	1.3	2.0	3.1	4.9	8.0
Barley	8.0	10.0	13.0	18.0	56.0

Source: adapted from Mass and Hoffman 1977; and Abrol (1973)

Salinity is known to depress the growth and symbiotic performance of nodulated legumes. Nodulation capacity and nodule function are adversely affected even under mild stress that otherwise would have no adverse effect on plant growth. Under salt stress, the symbiotic relationship between the bacteroids and host plant is adversely affected. Salinity tolerance is related to a higher nodulation capacity, which enables the maintenance of a higher level of nitrogen fixation in saline conditions. [54]

53 TJ Flowers, PM Gaur, CLL Gowda, L Krishnamurthy, S Samineni, KHM Siddique, NC Turner, V Vadez, RK Varshney, TD Colmer (2010) Salt sensitivity in chickpea. Plant, Cell & Environment, 33 (4), 490–509, http://onlinelibrary.wiley.com/doi/10.1111/j.1365-3040.2009.02051.x/full

54 B Singh, BK Singh, J Kumar, SS Yadav, K Usha (2005) Effects of salt stress on growth, nodulation, and nitrogen and carbon fixation of ten genetically diverse lines of chickpea (*Cicer arietinum* L.). Aust J Ag Research. 56 (5), 491–495.

IN FOCUS

Effects of salt stress on growth, nodulation, and nitrogen and carbon fixation

Researchers compared 10 genetically diverse chickpea lines for salt tolerance over two dry seasons. They studied growth, nodulation, moisture content, and nodule nitrogen and carbon fixation. The chickpea lines were raised in an open-air chamber in soil supplied with 0, 50, 75, and 100 mm NaCl. The shoot, root, and the single-plant weight declined as the level of salt increased. An almost identical pattern of response was observed for nodule number, weight per nodule, and nitrogen and carbon fixation. They found no distinct relationship among root/shoot ratio, plant moisture content, and salt-tolerance response of the chickpeas.

However, they did find that nodulation capacity (number and mass) under salt stress was related to salt-tolerance response. This trait could be used to improve salt tolerance of this legume species in order to increase its productivity and stability in saline soils. The researchers also demonstrated that nodule number, and not nodule mass, is the trait that can be used as a useful marker in studying salt stress in chickpeas. [55]

A 2016 study concluded that salt sensitivity in chickpeas is determined by Na(+) toxicity. [56] Another study suggested that Na+ accumulation in leaves is associated with delayed flowering, and this plays a role in the lower reproductive success of the sensitive lines. The delay is longer in sensitive genotypes than in tolerant ones. Filled pod and seed numbers, but not seed size, have been associated with reduced seed yield in saline conditions. [57]

It has also been found that salt stress reduces photosynthesis, decreases tissue sugars by 22–47%, and severely impairs vegetative and reproductive growth. [58]

Salt stress is thought to reduce germination either by making less water available for imbibition or by altering enzymatic activity, growth-regulator balance or protein metabolism in germinating seeds. One study has found that pre-soaking seeds for 24 hours in normal ground or tap water (0.8 dS m-1) increased germination by 27% compared to direct sowing. Sowing at depth of 4 cm also increased seedling growth under saline soils compared with sowing at 2 cm and 6 cm. [59]

Altered growth-hormone balance during germination is another factor resulting in poor germination and seedling growth under salt stress. The application of growth regulators such as gibberellic acid and kinetin have been found to increase germination (32%), root (32%) and shoot (153%) dry mass of seedlings stressed by salt.

55 B Singh, BK Singh, J Kumar, SS Yadav, K Usha (2005) Effects of salt stress on growth, nodulation, and nitrogen and carbon fixation of ten genetically diverse lines of chickpea (*Cicer arietinum* L.). Aust J Ag Research, 56 (5), 491–495

56 HA Khan, KHM Siddique, TD Colmer (2016) Salt sensitivity in chickpea is determined by sodium toxicity. Planta, 244 (3), 1–15, https://www.ncbi.nlm.nih.gov/pubmed/27114264

57 R Pushpavalli, J Quealy, TD Colmer, NC Turner, KHM Siddique, MV Rao, V Vadez (2016) Salt stress delayed flowering and reduced reproductive success of chickpea (*Cicer arietinum* L.), a response associated with Na+ accumulation in leaves. Journal of Agronomy and Crop Science, 202 (2), 125–138, http://onlinelibrary.wiley.com/doi/10.1111/jac.12128/abstract

58 HA Khan, KHM Siddique, TD Colmer (2016) Vegetative and reproductive growth of salt-stressed chickpea are carbon-limited: sucrose infusion at the reproductive stage improves salt tolerance. Journal of Experimental Botany. Published online May 2016. DOI 10.1093/jxb/erw177, http://jxb.oxfordjournals.org/content/early/2016/04/29/jxb.erw177.full

59 S Samineni (2010) Physiology, Genetics and QTL Mapping of Salt Tolerance in Chickpea. Thesis. University of Western Australia.

IN FOCUS

Physiological, chemical and growth responses to irrigation with saline water

Research in southern Morocco investigated the effect of irrigation with saline water on a local variety of chickpeas.

Results showed that, the more saline the irrigation water became, the less plants grew, and the less biomass they accumulated, and these led to reduced water uptake, water productivity and grain yield. In contrast, proline, soluble sugars, Na+ and Na+:K+ ratio increased as the irrigation water became more saline. The findings highlighted the role of proline and soluble sugars as osmolytes produced by chickpeas to mitigate the effect of salt stress.

Information can be used to determine threshold values. [60]

14.5.2 Soil chloride levels

Soil chloride levels >600 mg/kg have been found to reduce root growth in crops such as chickpeas, lentils and linseed. Soil analysis should be conducted to identify the level of chloride and at what depth it changes. Thresholds for chloride concentration in soil and yield reductions differ between crops (Tables 9 and 10).

Table 9: *Thresholds for chloride concentration in soil (mg/kg).*

Crop	10% yield reduction	50% yield reduction
Chickpeas	**600**	**1,000**
Bread wheat	700	1,500
Durum wheat	600	1,200
Barley	800	1,500
Canola	1,200	1,800

Source: Queensland Natural Resources and Water Bulletin

60 A Hirich, H El Omari, SE Jacobsen, N Lamaddalena, A Hamdy, R Ragab, A Jelloul, R Choukr-Allah (2014) Chickpea (*Cicer arietinum* L.) physiological, chemical and growth responses to irrigation with saline water. Australian Journal of Crop Science, 8 (5), 646–654, http://research.ku.dk/search/?pure=en%2Fpublications%2Fchickpea-cicer-arietinum-l-physiological-chemical-and-growth-responses-to-irrigation-with-saline-water(d1e5a11e-b2a9-4d12-b9dd-7ac6d9937dea).html

Table 10: *Soil constraint ratings for concentration of chloride (Cl) and sodium (Na).*

Low	Medium	High
Surface soil (top 10 cm)		
<300 mg Cl/kg	300–600 mg Cl/kg	>600 mg Cl/kg
<200 mg Na/kg	200–500 mg Na/kg	>500 mg Na/kg
Subsoil (10 cm to 1 m)		
<600 mg Cl/kg	600–1,200 mg Cl/kg	>1,200 mg Cl/kg
<500 mg Na/kg	500–1,000 mg Na/kg	>1,000 mg Na/kg

Source: Queensland Natural Resources and Water Bulletin

Agronomic practices and crop choices

Agronomic practices and crop choices may have to vary for differing levels of soil salinity or sodicity constraints. Pulses such as chickpeas can be grown only where there is low salinity.

Low constraints of Na and Cl (<600 mg Cl/kg, <500 mg Na/kg in top 1 m of soil):

- Cereal–legume rotations are possible.
- Canola can be grown.
- Opportunity cropping to utilise available soil water can be tried.

Medium constraints of Na and Cl (600–1,200 mg Cl/kg, 500–1,000 mg Na/kg in top 1 m of soil), growing tolerant crops such as wheat, barley and canola:

- Consider tolerant crop varieties.
- The more tolerant of the pulses (vetch, faba beans, and possibly lupins and field peas) will likely suffer yield reductions if grown.
- Match inputs to realistic yields.
- Cereal diseases must be managed.
- Avoid growing salt-susceptible pulses (i.e. lentils, chickpeas) or legumes, and durum wheat.
- Opportunity cropping to utilise available soil water can be tried, but options may be more limited.

High constraints of Na and Cl (>1,200 mg Cl/kg, >1,000 mg Na/kg in top 1 m of soil):

- Avoid growing crops, or grow tolerant cereals.
- Match inputs to realistic yields.
- Consider alternative land use to cropping (e.g. saline-tolerant forages, pastures).

14.5.3 Soil pH

Chickpea crops are best suited to well-drained loam and clay loam soils that are neutral to alkaline (pH 6.0 to 9.0). [61]

Acidic soils

Acid soils can significantly reduce production and profitability before paddock symptoms are noticed. Danger levels for crops are when soil pH is <5.5 (in $CaCl_2$) or 6.3 (in water). Monitor changes in soil pH by testing the soil regularly. If severe acidity is allowed to develop, irreversible soil damage can occur. Prevention is better than cure, so apply lime regularly in vulnerable soils. The most effective liming sources have a high neutralising value and a high proportion of material with a particle size <0.25 mm. More lime is required to raise pH in clays than in sands. Liming can induce manganese deficiency where soil manganese levels are already marginal. Low soil pH often leads to poor or ineffective nodulation in pulses because acid soil conditions affect initial numbers and multiplication of rhizobia. Field peas, faba

61 Agriculture Victoria.(2016). Growing Chickpea. http://agriculture.vic.gov.au/agriculture/grains-and-other-crops/crop-production/growing-chickpea

beans, lentils and chickpeas are vulnerable, as are vetches. Lupins are an exception because their rhizobia (Group G) are acid-tolerant. Granular inoculums seem to provide greater protection to rhizobia in acid soils.

Between pH Ca 5.5 and pHCa8 is the ideal pH range for plants. Soil pH targets are 5.5 in the topsoil, 0–10 cm, and >4.8 in the subsurface soil, 10 cm and below. At pH ca of 4.8 or lower, levels of aluminium in the soil become toxic. Free aluminium has a large impact on crop yield. It reduces root growth, which in turn reduces the depth of soil the plant has access to.

In terms of lime movement through the soil, a pH level of 5.5 is required in the top 0–10 cm of soil before lime can influence any soil deeper than this. Lime applied to the surface will be worked in with the traffic of the seeding implement. This creates a layer where the pH is ameliorated to the depth of the seeding point but no further. Lime must be in contact with the soil of low pH in order to react. This layering effect has an impact on yield potential of rotation crops and pastures. An ameliorated surface, above pH Ca 5.5, and subsurface with pH Ca below 4.8 reduces the yield potential of rotation crops and the efficacy of N fixation. After lime has been applied, the subsurface pH will remain unchanged until the lime is able to leach through the profile.

It is difficult to make the correct decisions on soil treatment and crop choice if you do not have full knowledge of the soil pH to depth. This is particularly so when the crop is susceptible to low pH or aluminium toxicity, as are break crops such as chickpeas. Poor yields of these rotation crops may be the result of low pH at depth, in spite of good pH at the surface.

Alkaline soils

Soil alkalinity is mainly caused by bicarbonates and carbonates, although phosphates, borates and some organic molecules can also contribute. In a soil with pH from 7 to 8.2, bicarbonates and carbonates of calcium and magnesium dominate.

Calcareous soils contain from 1–90% lime material as calcium carbonates, and these sparingly soluble salts cause the soil to have a pH of 8.0–8.2, which is not a severe problem for plant growth or agricultural production.

Problems are encountered in alkaline soils when sodium occurs or accumulates and forms salts such as sodium bicarbonate and sodium carbonate. These are highly soluble and increase the soil pH above 8.

When the pH is more than 9, the soils are considered to be highly alkaline and often have toxic amounts of bicarbonate, carbonate, aluminium and iron. The high amount of exchangeable sodium in these soils reduces soil physical fertility, and nutrient deficiency is also likely to be a major problem.

In alkaline soils, the abundance of carbonates and bicarbonates can reduce crop growth and induce nutrient deficiencies. The presence of free lime has a major impact on lupin growth, inducing iron and manganese deficiency, which cannot be corrected by foliar sprays of those nutrients.

Managing soil pH

- Growers are applying more lime per hectare than in the past but, in many cases, much more lime is needed to replenish the soil profile.
- Liming to remove soil acidity as a production constraint can also bring the benefits of increasing yields, increasing crop and pasture choice, and helping to protect the soil resource.

Soil acidification is an ongoing issue

Soil acidification is an ongoing and unavoidable result of productive agriculture. The main practices that cause soil acidification include the removing of products by harvest (Photo 14) and the leaching of nitrate from soil. Because soil acidification is an ongoing consequence of farming, management also needs to be ongoing.

Photo 14: *Hay production, especially legume hay, is one of the most acidifying practices.*

Source: Black Diamond Images, Flickr

Acid soils

Acid soils can be economically managed by the addition of agricultural lime, usually in the form of crushed limestone. Sufficient lime should be added to raise the pH to above 5.5. The amount of lime required to ameliorate acid soils will vary, depending mainly on the quality of the lime, the soil type and how acidic the soil has become.

Soils prone to becoming acidic will need liming every few years. Seek advice on an appropriate liming regime from your local agricultural adviser.

Alkaline soils

Treating alkaline soils by the addition of acidifying agents is not generally a feasible option due to the large buffering capacity of soils and uneconomic amounts of acidifying agent (e.g. sulfuric acid, elemental sulfur or pyrites) required.

Gypsum will reduce sodicity and this can reduce alkaline pH to some extent. Growing legumes in crop rotations may help in sustaining any reduction in pH.

In high-pH soils, using alkalinity-tolerant species and varieties of crops and pasture can reduce the impact of high pH. [62]

The importance of soil testing

Soil testing has a vital role in the management of soil acidity. It is important that decisions on where and when to apply lime and how much lime to apply are based on objective measures of soil acidity. While it is possible to estimate maintenance liming rates based on farm inputs and outputs, the most direct method, and the only way to measure existing acidity, is to regularly test the surface and subsurface soil layers.

Yield increases from liming

Liming can increase grain yield when soil pH is below recommended targets, and when soil pH is one of the factors constraining yield. Trials in WA found that, on average, applying lime increased yield by 0.2 t/ha, or 10%. This result is similar to what has been found in most other trials around Australia.

62 P Rengasamy (n.d.) Soil pH—South Australia. Fact sheet. Soilqualiy, http://www.soilquality.org.au/factsheets/soil-ph-south-austral

MORE INFORMATION

B Upjohn, G Fenton, M Conyers (2005) Soil acidity and liming. 3rd edn. Agdex 534. NSW Department of Primary Industries.

However, the yield increases from liming may be even higher than this. When trials have included ripping or tilling, increases in yield have been even greater. Also, it takes a few years for lime to react with the soil and increase the soil pH. When yield was calculated starting from the third harvest after lime was applied, the average yield increase was 0.25 t/ha, or 16%.

Yield and yield gains from liming will depend on the relationship between paddock yield and yield potential. If the paddock yield is low relative to the yield potential there are likely to be additional constraints present, and there may be little gain from liming. If the paddock yield is already close to potential, pH is not likely to be a constraint and there is little immediate gain to be made from liming.

However, maintenance liming will be required to counter ongoing acidification and maintain the productivity of the paddock. [63]

14.5.4 Sodicity

- Sodic soils occupy almost one-third of the land area of Australia.
- Sodicity has serious impacts on farm production, as well as significant off-site consequences such as:
- surface crusting
- reduced seedling emergence
- reduced soil aeration
- increased risk of run-off and erosion
- less groundcover and organic matter
- less microbial activity
- Sodic soils are known as dispersive clays and reduce seedling emergence (Photo 15).
- Sodic soils are can lead to tunnel erosion—they turn to slurry when wet, and channels are easily created through them by moving water. [64]

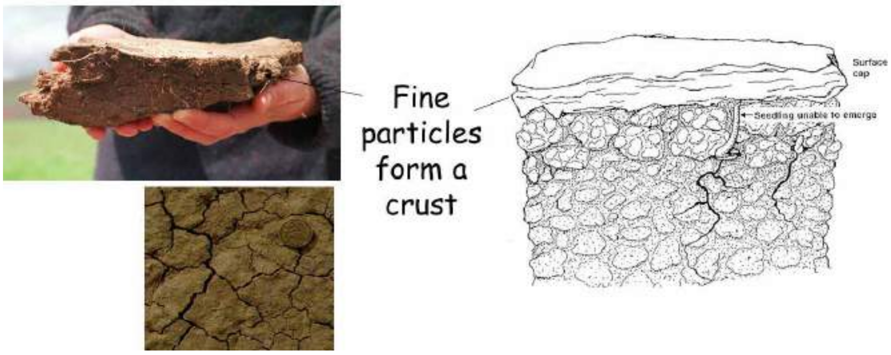


Photo 15: *The impact of sodic soils on seedling emergence: surface crusting and dispersive clay limit emergence.*

Source: Corangamite CMA

Chickpeas are classified among the most sensitive of all field crops to sodic soil conditions.

Soils high in sodium are structurally unstable, with clay particles dispersing when wet. This subsequently blocks soil pores, which reduces water infiltration and aeration, and retards root growth. On drying, a sodic soil becomes dense and forms a hard surface crust up to 10 mm thick. This can restrict seedling emergence. Some indicators of surface sodicity include:

- soils being prone to crusting and sealing up

63 C Grazey, J Carson (n.d.) Managing soil acidity—Western Australia Fact sheet. Soilquality, http://www.soilquality.org.au/factsheets/managing-soil-acidity-western-australia

64 Corangamite CMA (2013) How do I manage the impact of sodic soils? Corangamite CMA, http://www.ccmaknowledgebase.vic.gov.au/brown_book/04_Sodic.htm

- ongoing problems with poor plant establishment
- the presence of scalded areas in adjoining pasture

Exchangeable sodium percentage is the measure for sodicity, and soils are rated thus:

- ESP <3: non-sodic soils
- ESP 3–14: sodic soil
- ESP >15: strongly sodic

Soils that are sodic in the topsoil have the greatest impact on crop performance (see Figure 8 for effect of ESP on chickpea yield). Sodic layers deeper in the soil profile are not as great a concern but can still affect yields by restricting root development and water extraction. The net effect of severely restricted root growth in chickpeas is usually the early onset of drought stress.

It is unlikely that soil sodic layers deeper than 90 cm will have significant impact on chickpea yields. [65]

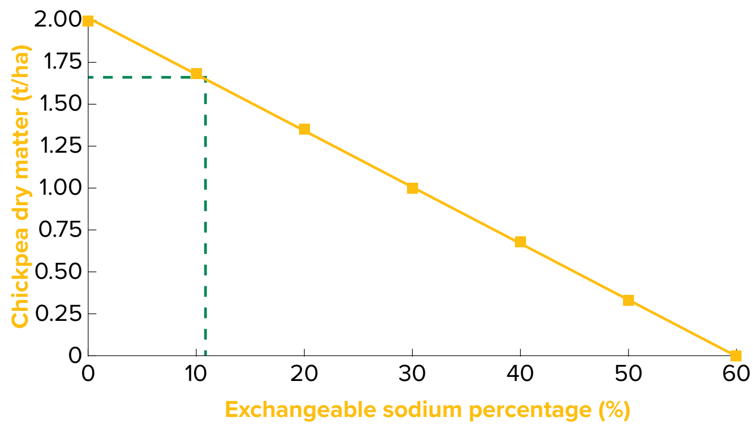


Figure 8: *Impact of sodicity on the production of chickpea dry matter.*

Source: Pulse Australia

Managing sodic soils

- Growers need to correctly identify the problem first and ensure that the soils are in fact sodic.
- Sodic soils can be directly treated through the application of gypsum (particularly on the surface), which serves to replace the excess sodium in the soil with calcium.
- In southern Victoria, typical application rates of gypsum are around 2.5 t/ha and applied every 3–5 years.
- The application of lime to sodic soils acts in a similar manner to gypsum, but is much slower acting and less effective.
- Although the application of gypsum can effectively counter sodicity in the short run, longer-term management strategies need to be in place to increase, and then maintain, organic matter in soils. Increased organic matter can improve hard-setting soils, and it can also enhance the effect of gypsum.
- Sodicity can also be reduced by maintaining adequate vegetation cover, leaf litter or stubble on the soil surface.
- Trials in the high-rainfall zone of southern Victoria have shown that the amelioration of dense sodic subsoil using organic amendments can increase wheat yield more than using gypsum. [66]

65 Pulse Australia (2013) Northern Chickpea—Best Management Practices Training Course Manual. Pulse Australia.

66 P Sale, J Gill, R Peries, C Tang (2008) Amelioration of dense sodic subsoil using organic amendments increases wheat yield more than using gypsum in a high rainfall zone of southern Australia. La Trobe University, Bundoora, Victoria.

Marketing

The final step in generating farm income is converting the tonnes produced into dollars at the farm gate. This section provides best-in-class marketing guidelines for managing price variability to protect income and cash flow.

Figure 1 shows a grain selling flow chart that summarises:

- The decisions to be made.
- The drivers behind the decisions.
- The guiding principles for each decision point.

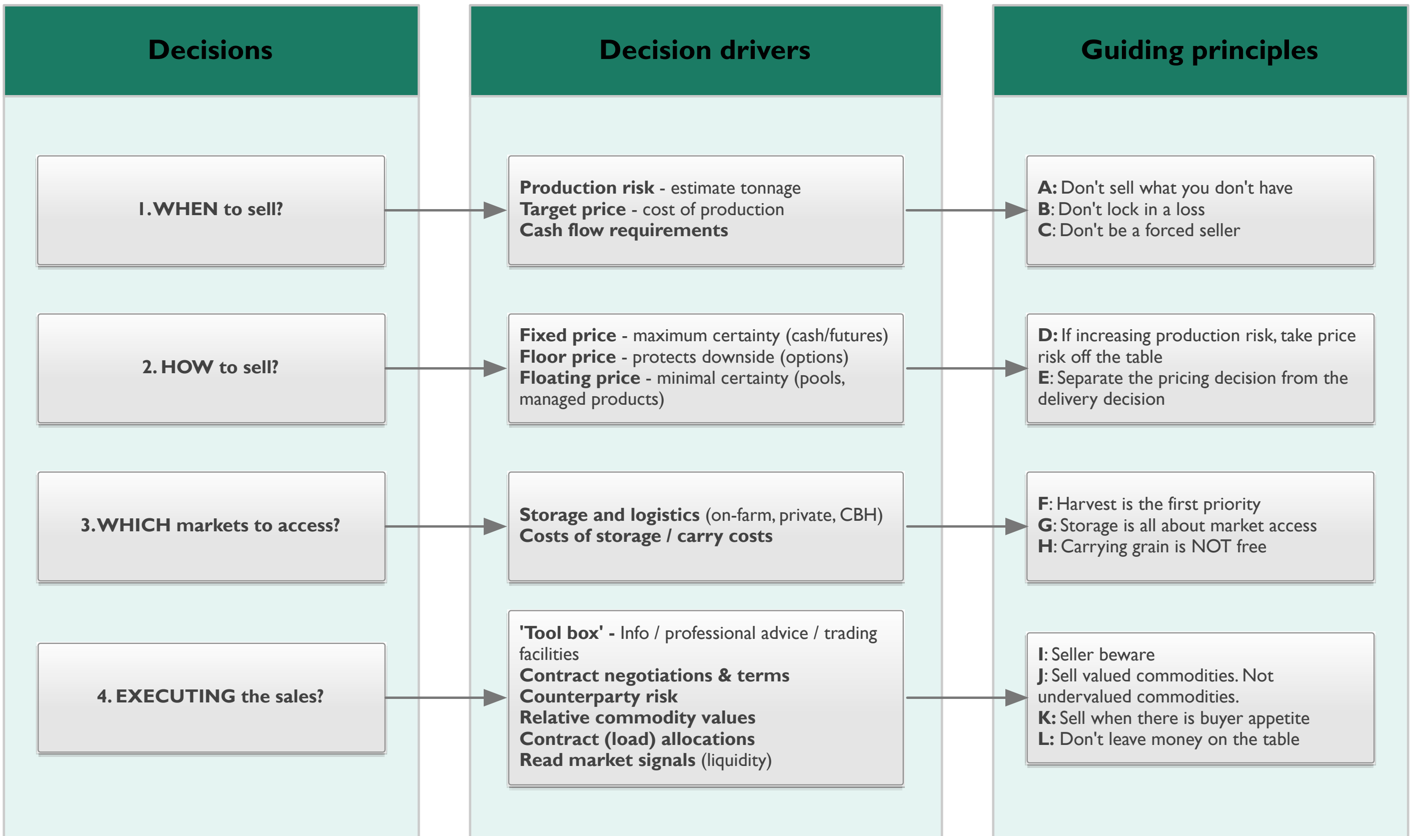
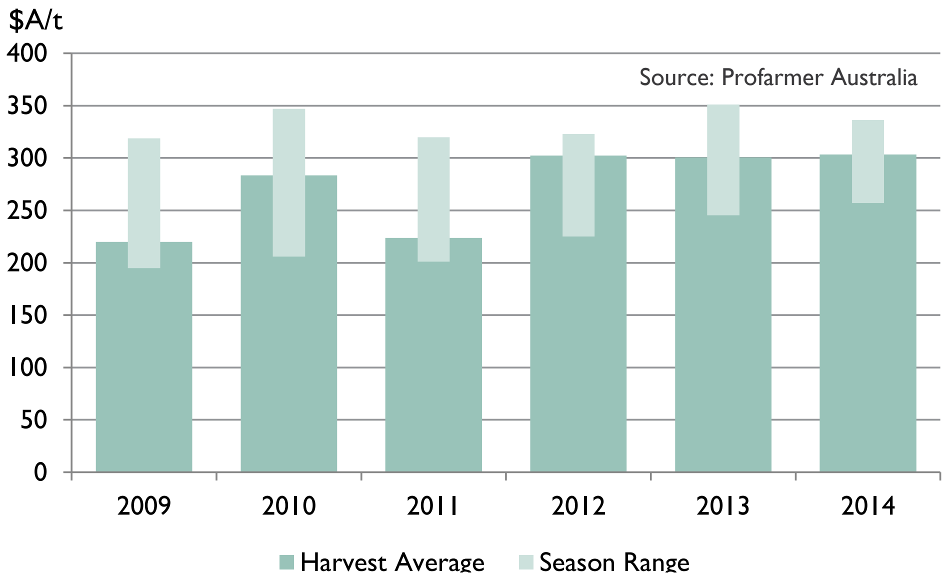


Figure 1: *Grain-selling flowchart.*

The grower will run through a decision-making process each season, because growing and harvesting conditions, and prices for grains, change all the time. For example, in the six years to and including 2014, Newcastle APWI wheat prices varied A$70–$150/t, a variability of 25–60% (Figure 2). For a property producing 1,000 tonnes of wheat this means $70,000–$150,000 difference income, depending on timing of sales.



Note to figure:
Newcastle APW1 wheat prices have varied A$70-$150/t over the past 6 years (25-60% variability). For a property producing 1,000 tonne of wheat this means $70,000-$150,000 difference in income depending on price management skill.

Figure 2: *Newcastle APWI price variation, 2009–2014.*

Source: Profarmer Australia

15.1 Selling principles

The aim of a selling program is to achieve a profitable average price (the target price) across the entire business. This requires managing several unknowns to establish a target price and then work towards achieving the target price.

Unknowns include the amount of grain available to sell (production variability), the final cost of producing the grain, and the future prices that may result. Australian farm-gate prices are subject to volatility caused by a range of global factors that are beyond our control and are difficult to predict.

The skills growers have developed to manage production unknowns can also be used to manage pricing unknowns. This guide will help growers manage and overcome price uncertainty.

15.1.1 Be prepared

Being prepared by having a selling plan is essential for managing uncertainty. The steps involved are forming a selling strategy, and forming a plan for effectively executing sales. The selling strategy consists of when and how to sell.

When to sell

Knowing when to sell requires an understanding of the farm's internal business factors, including:

- production risk
- a target price based on the cost of production and the desired profit margin
- business cashflow requirements

How to sell

Working out how to sell your grain is more dependent on external market factors, including:

- the time of year, which determines the pricing method
- market access, which determines where to sell
- relative value, which determines what to sell

The following diagram (Figure 3) lists the key principles to employ when considering sales during the growing season. Exactly when each principle comes into play is indicated in the discussion below of the steps involved in marketing and selling.

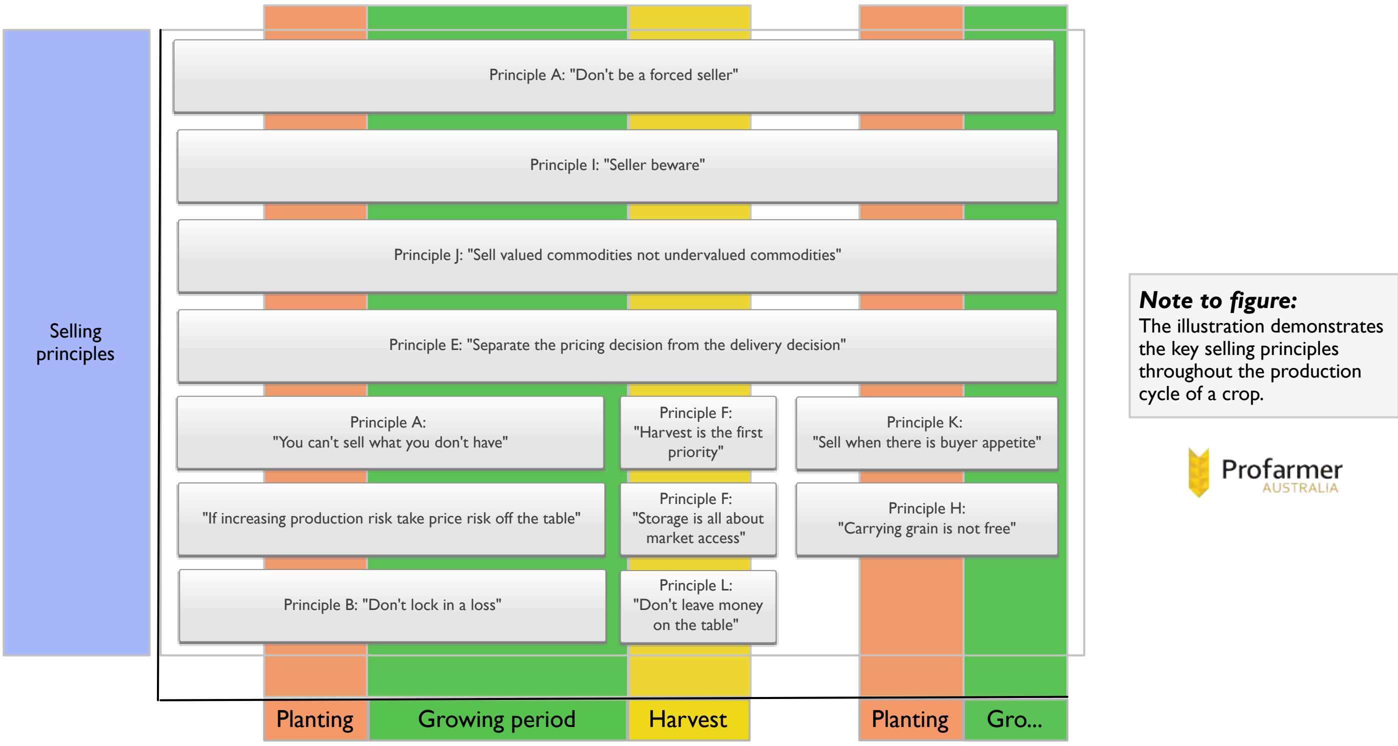


Figure 3: *Timeline of grower commodity selling principles.*

Source: Profarmer Australia

15.1.2 Establish the business risk profile

Establishing your business risk profile helps you determine when to sell: it allows you to develop target price ranges for each commodity, and provides confidence to sell when the opportunity arises. Typical business circumstances and how to quantify the risks during the production cycle are described below (Figure 4).

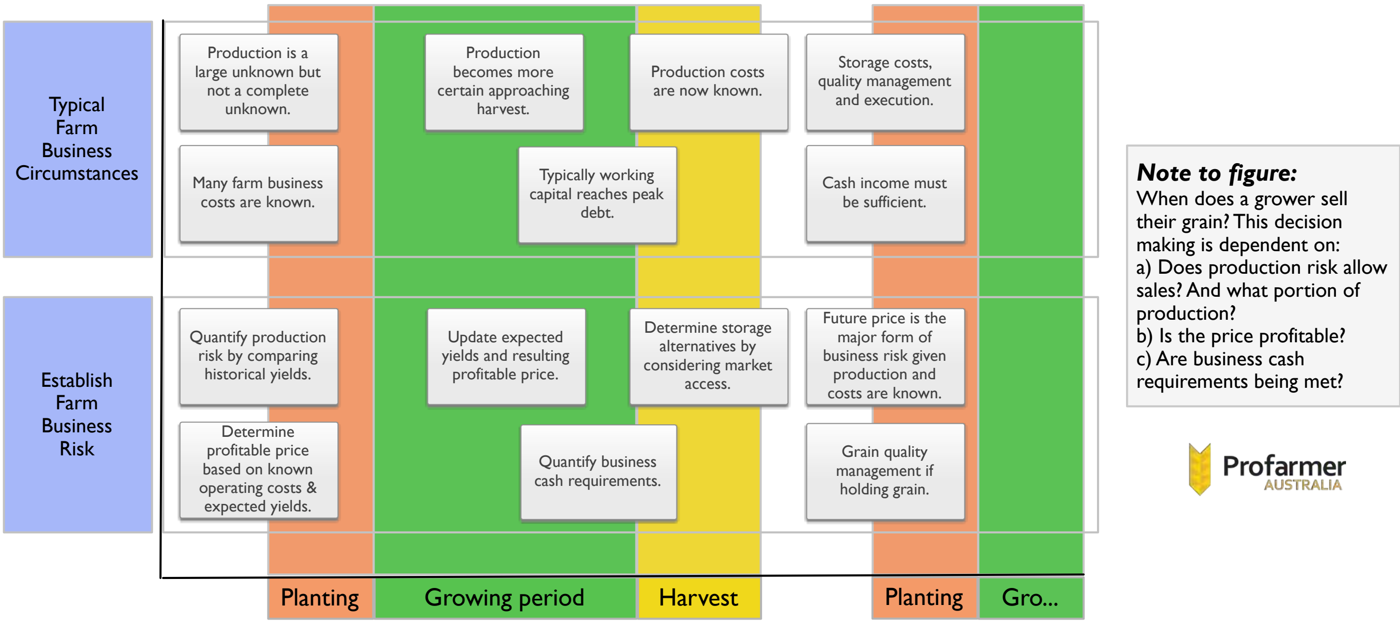


Figure 4: *Typical farm business circumstances and risk.*

Source: Profarmer Australia

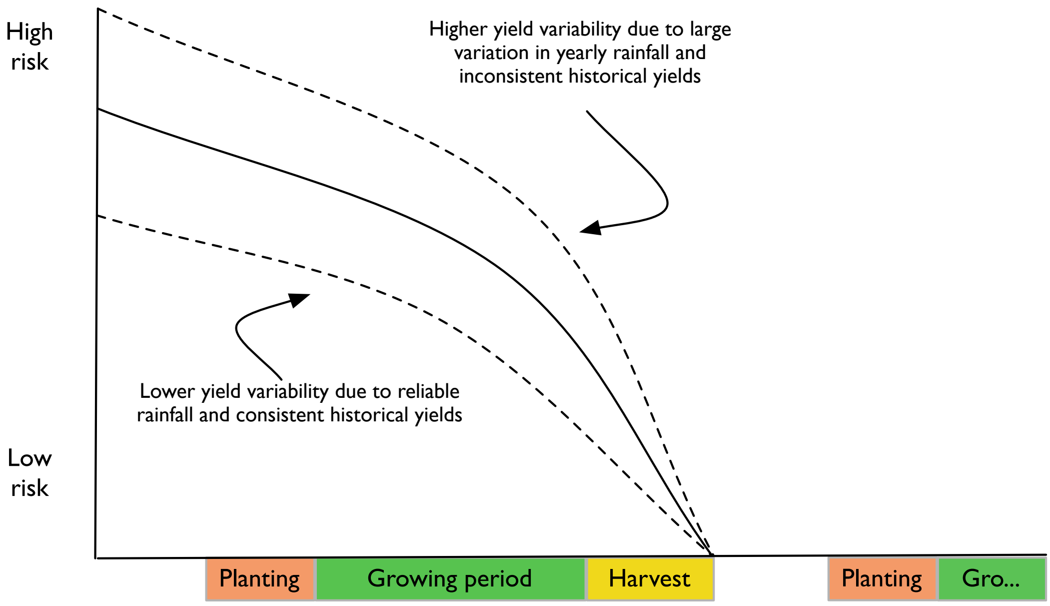
Production risk profile of the farm

Production risk is the level of certainty around producing a crop and is influenced by location (climate, season and soil type), crop type, crop management, and the time of the year.

Principle A: You can't sell what you don't have.

Therefore, don't increase business risk by over committing production. Establish a production risk profile (Figure 5) by:

1. Collating historical average yields for each crop type and a below-average and above-average range.
2. Assessing the likelihood of achieving the average, based on recent seasonal conditions and the seasonal outlook.
3. Revising production outlooks as the season progresses.



Note to figure:
The quantity of crop grown is a large unknown early in the year however not a complete unknown. 'You can't sell what you don't have' but it is important to compare historical yields to get a true indication of production risk. This risk reduces as the season progresses and yield becomes more certain. Businesses will face varying production risk levels at any given point in time with consideration to rainfall, yield potential, soil type, commodity etc.

Figure 5: *Typical risk profile of a farm operation.*

Source: Profarmer Australia

Establishing a target price

A profitable commodity target price is the cost of production per tonne plus a desired profit margin. It is essential to know the cost of production per tonne for the farm business, which means knowing all farming costs, both variable and fixed.

Principle B: Don't lock in a loss.

If committing production ahead of harvest, ensure the price will be profitable. The steps needed to calculate an estimated profitable price is based on the total cost of production and a range of yield scenarios, as provided below (Figure 6).

Estimating cost of production	
Planted Area	1,200 ha
Estimate Yield	2.85 t/ha
Estimated Production	3,420 t
Fixed costs	
Insurance and General Expenses	$100,000
Finance	$80,000
Depreciation/Capital Replacement	$70,000
Drawings	$60,000
Other	$30,000
Variable costs	
Seed and sowing	$48,000
Fertiliser and application	$156,000
Herbicide and application	$78,000
Insect/fungicide and application	$36,000
Harvest costs	$48,000
Crop insurance	$18,000
Total fixed and variable costs	$724,000
Per Tonne Equivalent (Total costs + Estimated production)	$212 /t
Per tonne costs	
Levies	$3 /t
Cartage	$12 /t
Receival fees	$11 /t
Freight to Port	$22 /t
Total per tonne costs	$48 /t
Cost of production Port FIS equiv	$259.20
Target profit (ie 20%)	$52.00
Target price (port equiv)	**$311.20**

Step 1: Estimate your production potential. The more uncertain your production is, the more conservative the yield estimate should be. As yield falls, your cost of production per tonne will rise.

Step 2: Attribute your fixed farm business costs. In this instance if 1,200 ha reflects 1/3 of the farm enterprise, we have attributed 1/3 fixed costs. There are a number of methods for doing this (see M Krause "Farming your Business") but the most important thing is that in the end all costs are accounted for.

Step 3: Calculate all the variable costs attributed to producing that crop. This can also be expressed as $ per ha x planted area.

Step 4: Add together fixed and variable costs and divide by estimated production

Step 5: Add on the "per tonne" costs like levies and freight.

Step 6: Add the "per tonne" costs to the fixed and variable per tonne costs calculated at step 4.

Step 7: Add a desired profit margin to arrive at the port equivalent target profitable price.

Figure 6: *An example of how to estimate the costs of production.*

Income requirements

Understanding farm business cash-flow requirements and peak cash debt enables growers to time grain sales so that cash is available when required. This prevents having to sell grain below the target price to satisfy a need for cash.

Principle C: Don't be a forced seller.

Be ahead of cash requirements to avoid selling in unfavourable markets.

Typical cash-flow to grow a crop are illustrated below (Figures 7 and 8). Costs are incurred up front and during the growing season, with peak working capital debt incurred at or before harvest. Patterns will vary depending on circumstance and enterprise mix. The second figure demonstrates how managing sales can change the farm's cash balance.

MORE INFORMATION

GRDC's manual Farming the Business also provides a cost-of-production template and tips on grain selling v. grain marketing.

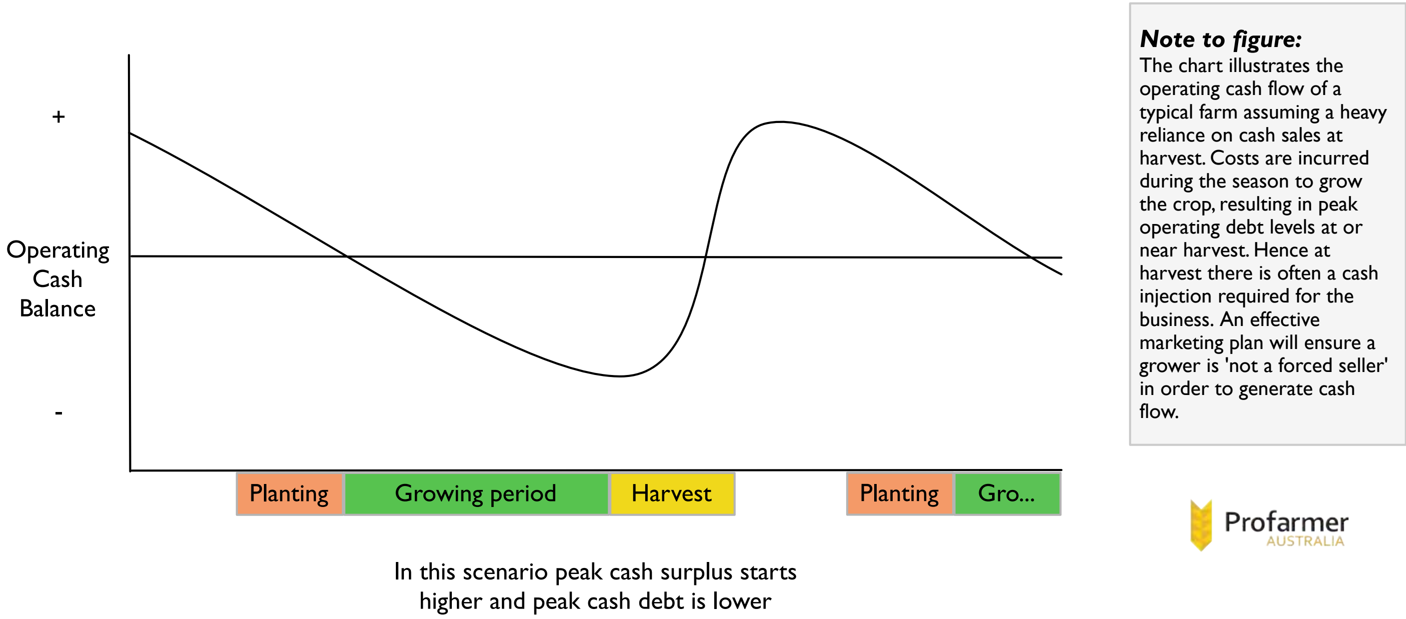


Figure 7: *A typical operating cash balance when relying on cash sales at harvest.*

Source: Profarmer Australia

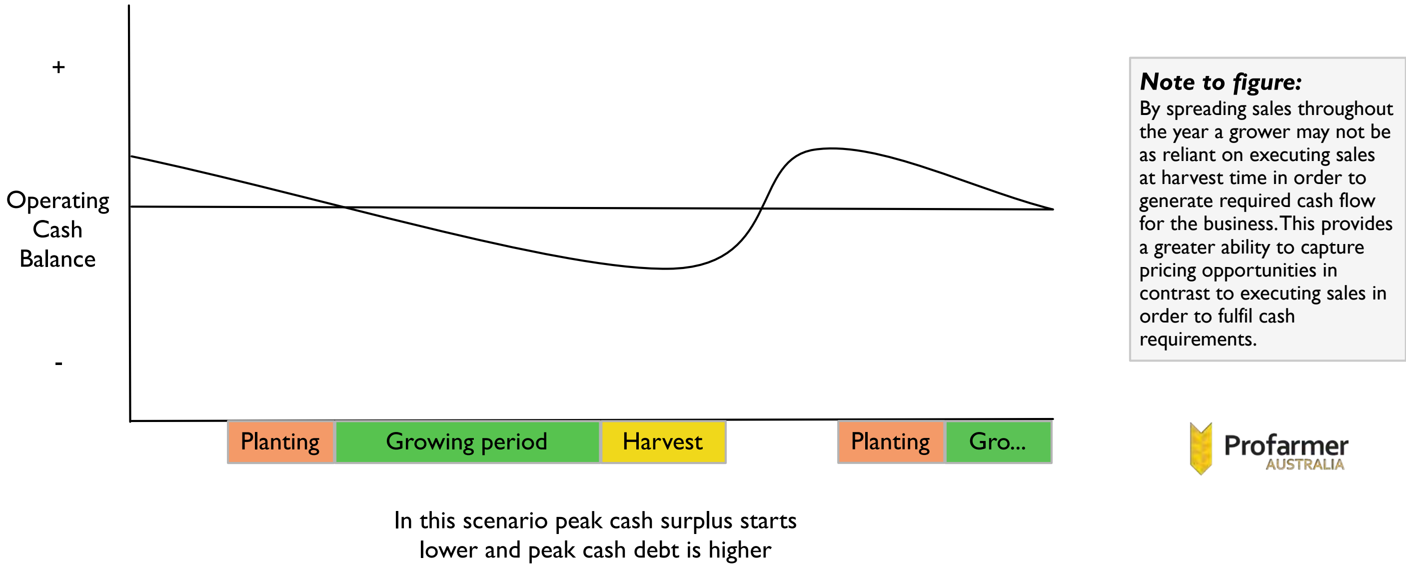


Figure 8: *Typical operating cash balance when crop sales are spread over the year.*

Source: Profarmer Australia

The when-to-sell steps above result in an estimated production tonnage and the risk associated with producing that tonnage, a target price range for each commodity, and the time of year when cash is most needed.

15.1.3 Managing your price

The first part of the selling strategy answers the question about when to sell and establishes comfort around selling a portion of the harvest.

The second part of the strategy, managing your price, addresses how to sell your crop.

Methods of price management

Pricing products provide varying levels of price risk coverage, but not all products are available for all crops (Table 1).

Table 1: *Pricing methods and how they are used for different crops.*

	Description	Wheat	Barley	Canola	Oats	Lupins	Field Peas	Chick Peas
Fixed price products	Provides the most price certainty	Cash, futures, bank swaps	Cash, futures, bank swaps	Cash, futures, bank swaps	Cash	Cash	Cash	Cash
Floor price products	Limits price downside but provides exposure to future price upside	Options on futures, floor price pools	Options on futures	Options on futures	none	none	none	none
Floating price products	Subject to both price upside and downside	Pools	Pools	Pools	Pools	Pools	Pools	Pools

Figure 9 summarises how the different methods of price management are suited to the majority of farm businesses.

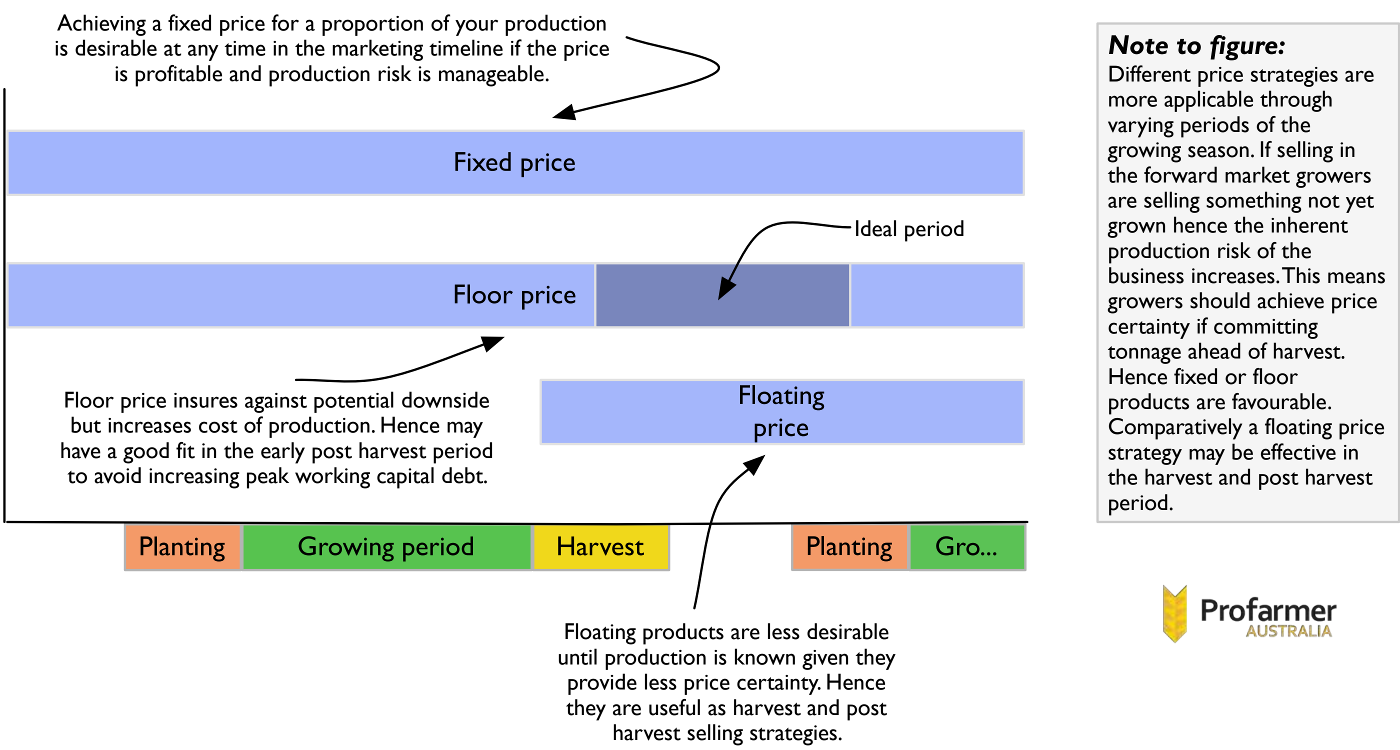


Figure 9: *Price strategy timeline, summarising the suitability for most farm businesses of different methods of price management for different phases of production.*

Source: Profarmer Australia

Principle D: If increasing production risk, take price risk off the table.

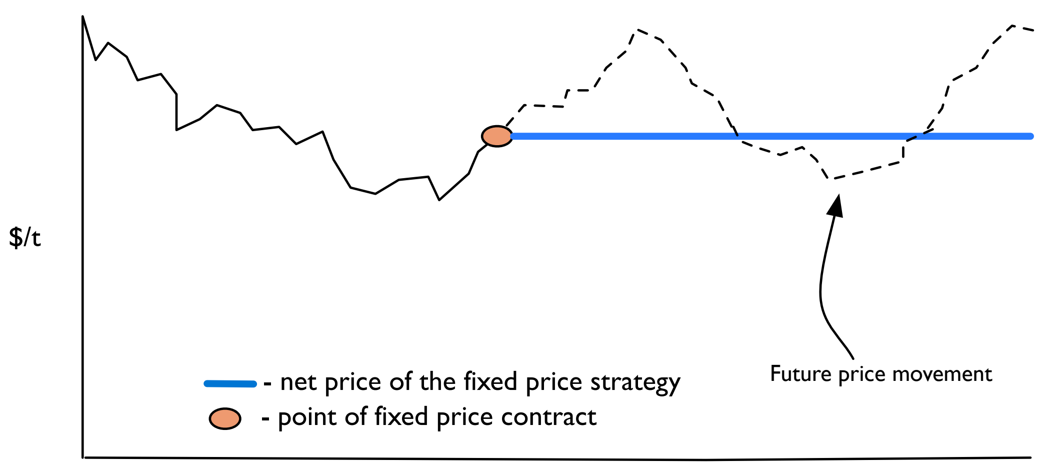
When committing to unknown production, price certainty should be achieved to avoid increasing overall business risk.

Principle E: Separate the pricing decision from the delivery decision.

Most commodities can be sold at any time with delivery timeframes being negotiable, hence price management is not determined by delivery.

Fixed price

A fixed price is achieved via cash sales and/or selling a futures position (swaps) (Figure 10). It provides some certainty around expected revenue from a sale as the price is largely a known factor, except when there is a floating component in the price, e.g. a multi-grade cash contract with floating spreads or a floating-basis component on futures positions.



Note to figure:
Fixed price product locks in price and provides certainty over what revenue will be generated regardless of future price movement.

Profarmer AUSTRALIA

Figure 10: *Fixed price strategy.*

Source: Profarmer Australia

Floor price

Floor-price strategies (Figure 11) can be achieved by utilising options on a relevant futures exchange (if one exists), or via a managed-sales program (i.e. a pool with a defined floor-price strategy) offered by a third party. This pricing method protects against potential future downside while capturing any upside. The disadvantage is that this kind of price 'insurance' has a cost, which adds to the farm's cost of production.

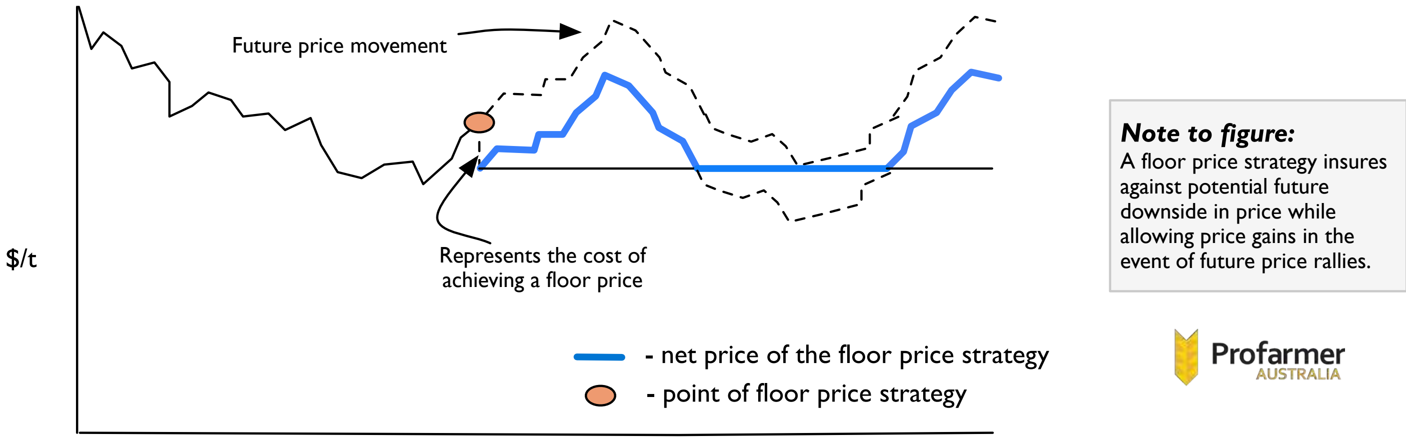


Figure 11: *Floor price strategy.*

Source: Profarmer Australia

3. Floating price

Many of the pools or managed-sales programs are a floating price, where the net price received will move up and down with the future movement in price (Figure 12). Floating-price products provide the least price certainty and are best suited for use at or after harvest rather than before harvest.

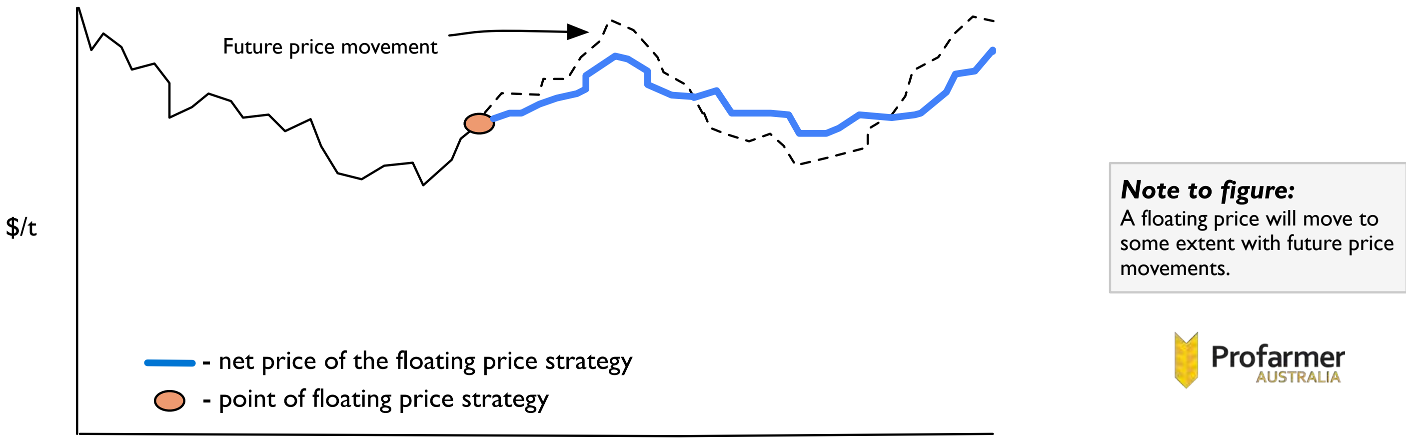


Figure 12: *Floating price strategy.*

Source: Profarmer Australia

Having considered the variables of production for the crop to be sold, and how these fit against the different pricing mechanisms, the farmer may revise their selling strategy, taking the risks associated with each mechanism into account.

Fixed-price strategies include physical cash sales or futures products, and provide the most price certainty, but production risk must be considered.

Floor-price strategies include options or floor-price pools. They provide a minimum price with upside potential and rely less on production certainty, but cost more.

Floating-price strategies provide minimal price certainty, and so are best used after harvest.

15.1.4 Ensuring access to markets

Once the questions of when and how to sell are sorted out, planning moves to the storage and delivery of commodities to ensure timely access to markets and execution of sales. Planning where to store the commodity is an important component of ensuring the type of access to the market that is likely to yield the highest return (Figure 13).

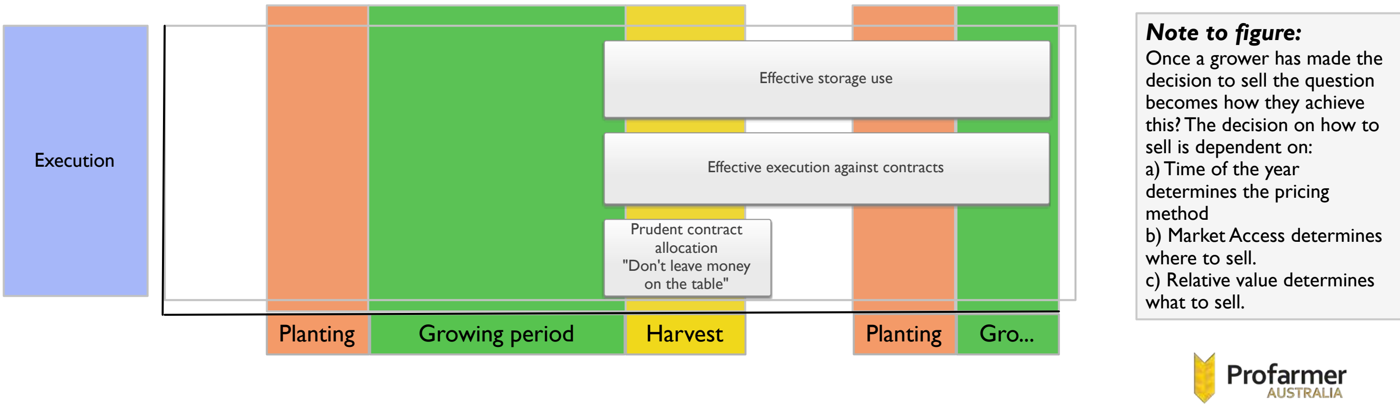


Figure 13: *Storage decisions are influenced by selling decisions and the timing of all farming activities.*

Source: Profarmer Australia

Storage and logistics

The return on investment from grain handling and storage expenses is optimised when storage is considered in light of market access so as to maximise returns as well as harvest logistics.

Storage alternatives include variations of bulk handling, private off-farm storage, and on-farm storage. Delivery and quality management are key considerations in deciding where to store your commodity (Figure 14).

Principle F: Harvest is the first priority.

During harvest, getting the crop into the bin is the most critical aspect of business success; hence storage, sale and delivery of grain should be planned well ahead of harvest to allow the grower to focus on the harvest itself.

Bulk export commodities requiring significant quality management are best suited to the bulk-handling system. Commodities destined for the domestic end-user market, (e.g. feedlot, processor, or container packer), may be more suited to on-farm or private storage to increase delivery flexibility.

Storing commodities on the farm requires prudent quality management to ensure that the grain is delivered to the agreed specifications. If not well planned and carried out, it can expose the business to high risk. Penalties for out-of-specification grain arriving at a buyer's weighbridge can be expensive, as the buyer has no obligation to accept it. This means the grower may have to incur the cost of taking the load elsewhere, and may also have to find a new buyer.

On-farm storage also requires that delivery is managed to ensure that the buyer receives the commodities on time and with appropriate weighbridge and sampling tickets.

Principle G: Storage is all about market access.

Storage decisions depend on quality management and expected markets.

MORE INFORMATION

For more information on on-farm storage alternatives and economics refer to Section 13: Grain Storage.

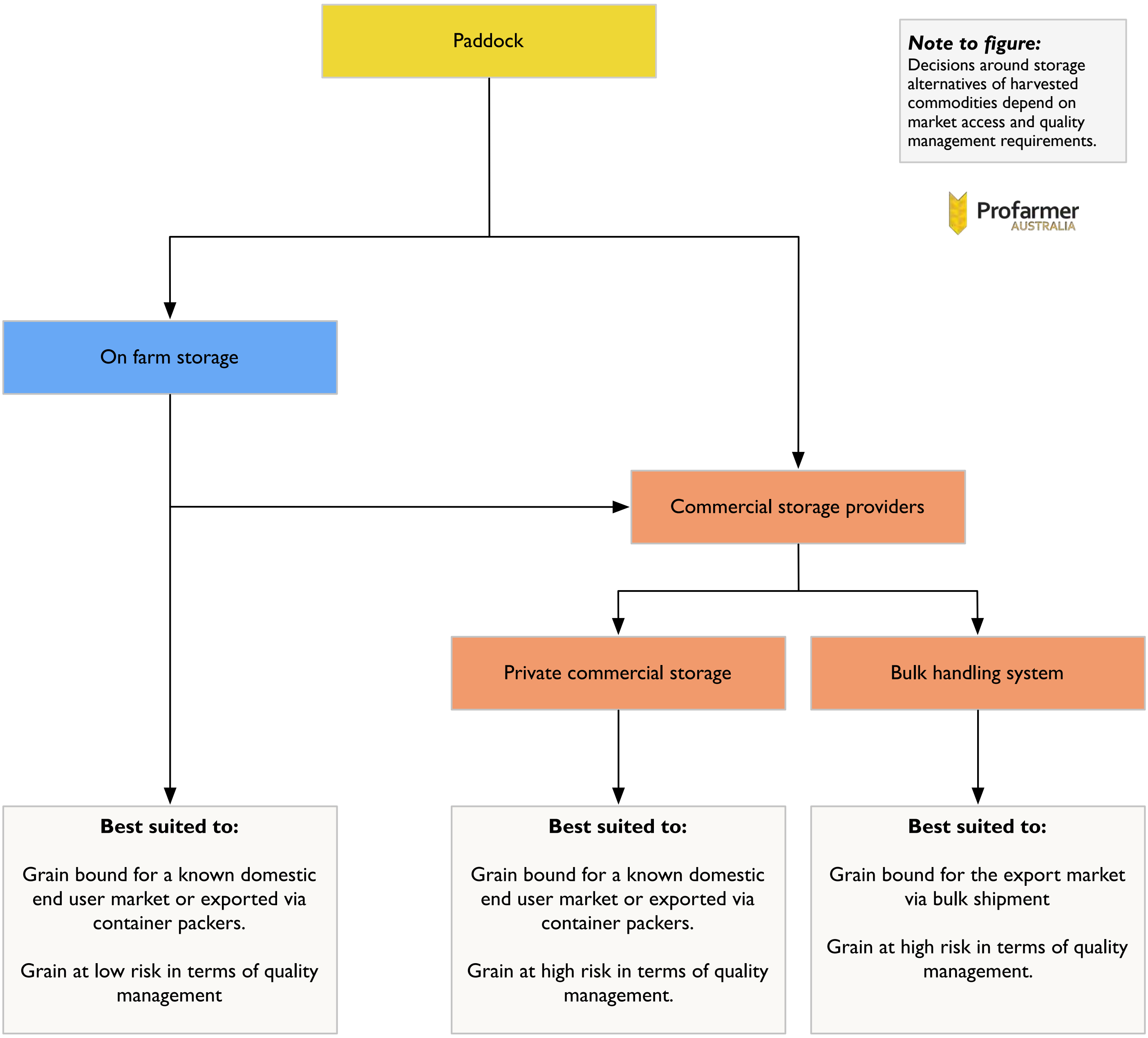


Figure 14: *Grain storage decision-making.*

Source: Profarmer Australia

Cost of carrying grain

Storing grain to access sales opportunities post-harvest invokes a cost to 'carry', or hold, the grain. Price targets for carried grain need to account for the cost of carrying it. Carrying costs are typically $3–4/t per month and consist of:

- Monthly storage fee charged by a commercial provider (typically ~$1.50–2.00/t).
- Monthly interest associated with having wealth tied up in grain rather than available as cash or for paying off debt (~$1.50–$2.00/t, depending on the price of the commodity and interest rates).

The price of carried grain therefore needs to be $3–4/t per month higher than the price offered at harvest (Figure 15).

The cost of carrying also applies to grain stored on the farm, as there is the cost of the capital invested in the farm storage plus the interest component. A reasonable assumption is a cost of $3–4/t per month for on-farm storage.

Principle H: Carrying grain is not free.

The cost of carrying grain needs to be accounted for if holding it for sale after harvest is part of the selling strategy. if selling a cash contract with deferred delivery, a carry charge can be negotiated into the contract. For example, a crop sold in March for delivery in March–June on the buyer's call at $700/t + $5/t per month carrying would generate an income of $715/t if delivered in June (Figure 15).

Figure 15: *Cash values compared with cash values adjusted for the cost of carrying.*

Source: Profarmer Australia

Optimising farm-gate returns involves planning the appropriate storage strategy for each commodity so as to improve market access and ensure that carrying costs are covered in the price received.

15.1.5 Converting tonnes into cash

This section provides guidelines for converting the selling and storage strategy into cash by effective execution of sales.

Set up the toolbox

Selling opportunities can be captured when they arise by assembling the necessary tools in advance. The toolbox for converting tonnes of grain into cash includes the following.

1. Timely information—this is critical for awareness of selling opportunities and includes:

- Market information provided by independent parties.
- Effective price discovery including indicative bids, firm bids and trade prices.
- Other market information pertinent to the particular commodity.

2. Professional services—grain-selling professional services and cost structures vary considerably. An effective grain-selling professional will put their clients' best interests first by not having conflicts of interest and by investing time in the relationship. A better return on investment for the farm business is achieved through higher farm-gate prices, which are obtained by accessing timely information, and being able to exploit the seller's greater market knowledge and greater market access.

MORE INFORMATION

Access to buyers, brokers, agents, products and banks through Grain Trade Australia

Commodity futures brokers

ASX, Find a futures broker

3. Futures account and a bank-swap facility—these accounts provide access to global futures markets. Hedging futures markets is not for everyone; however, strategies which utilise exchanges such as the Chicago Board of Trade (CBOT) can add significant value.

How to sell for cash

Like any market transaction, a cash–grain transaction occurs when a bid by the buyer is matched by an offer from the seller. Cash contracts are made up of the following components, with each component requiring a level of risk management (Figure 16):

- Price—future price is largely unpredictable, so devising a selling plan to put current prices into the context of the farm business is critical to managing price risk.
- Quantity and quality—when entering a cash contract, you are committing to deliver the nominated amount of grain at the quality specified, so production and quality risks must be managed.
- Delivery terms—the timing of the title transfer from the grower to the buyer is agreed at time of contracting. If this requires delivery direct to end-users, it relies on prudent execution management to ensure delivery within the contracted period.
- Payment terms—in Australia, the traditional method of contracting requires title on the grain to be transferred ahead of payment, so counterparty risk must be managed.

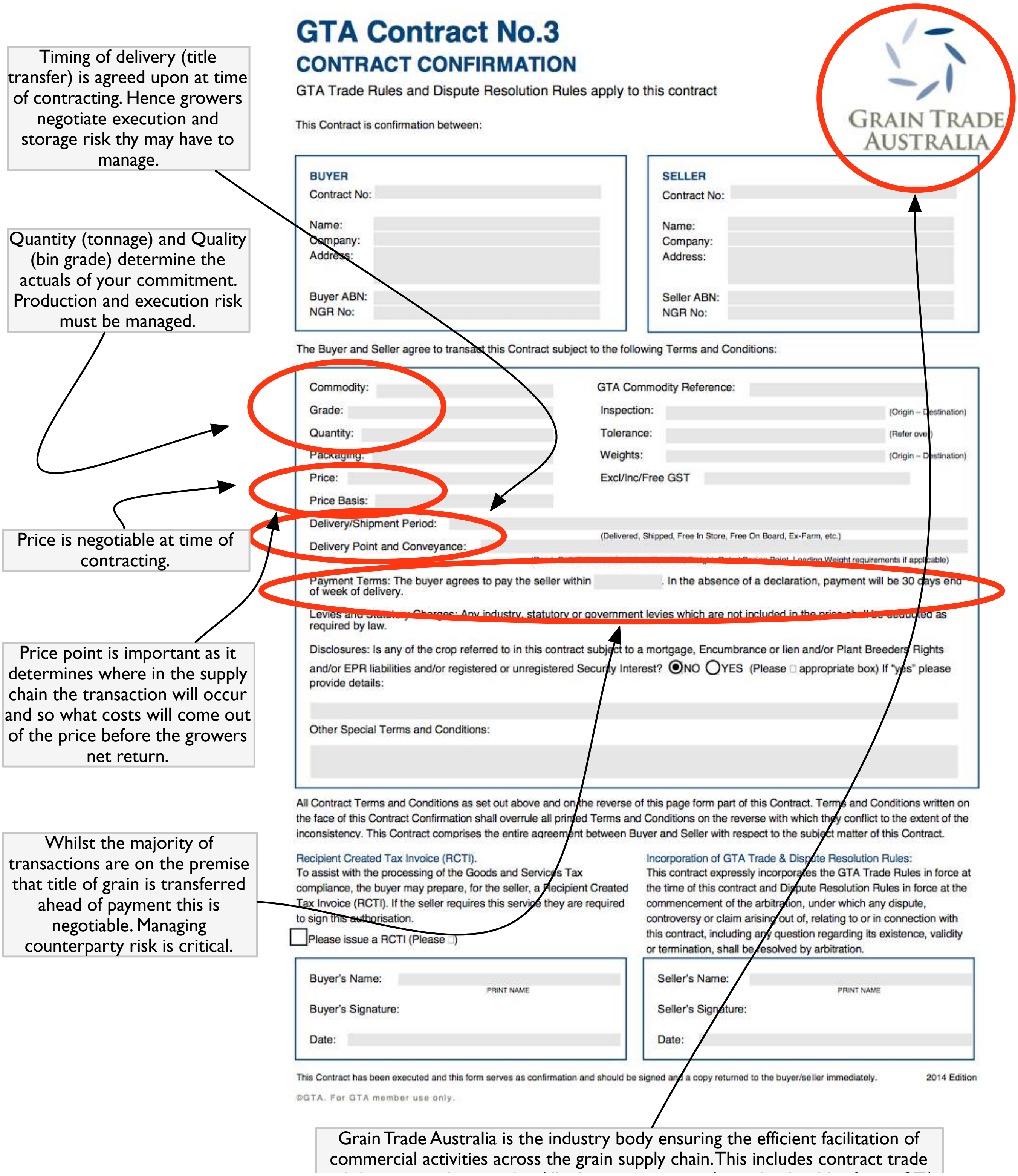


Figure 16: *Typical terms of a cash contract.*

Source: Grain Trade Australia

The price point within a cash contract will depend on where the transfer of grain title will occur along the supply chain. Figure 17 depicts the terminology used to describe these points and the associated costs to come out of each price before growers receive their net return.

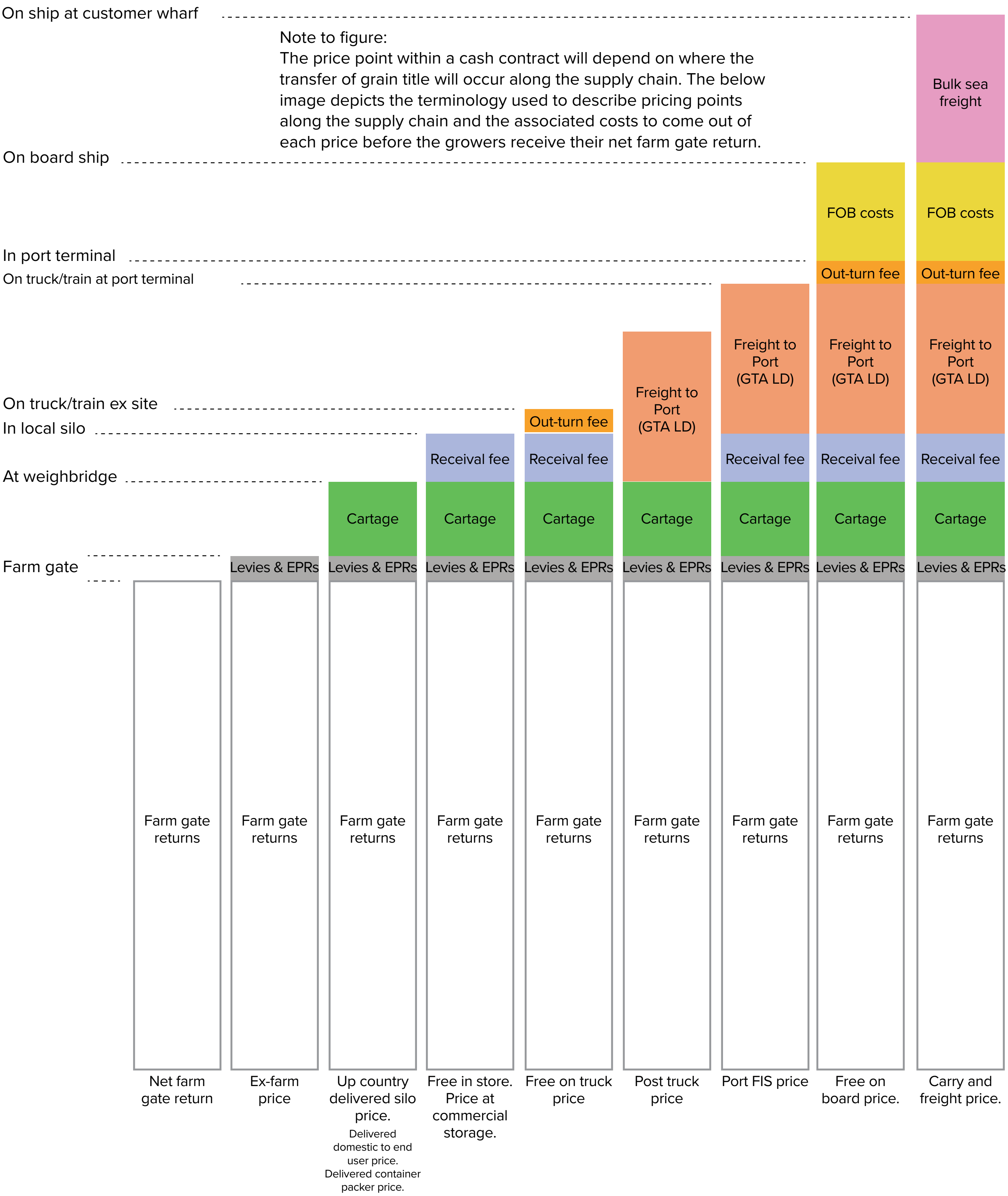


Figure 17: *Cost and pricing points throughout the supply chains.*

Source: Profarmer Australia

MORE INFORMATION

Grain Trade Australia, A guide to taking out grain contracts

Grain Trade Australia, Trading standards

GrainTransact Resource Centre

GrainFlow

Emerald Grain

Clear Grain Exchange, Getting started

Clear Grain Exchange, Terms and conditions

MORE INFORMATION

GTA, Managing counterparty risk

Clear Grain Exchange's title transfer model

GrainGrowers, Managing risk in grain contracts

Leo Delahunty, Counterparty risk: A producer's perspective

Cash sales generally occur through three methods:

- Negotiation via personal contact—traditionally prices are posted as a public indicative bid. The bid is then accepted or negotiated by a grower with the merchant or via an intermediary. This method is the most common and is available for all commodities.
- Accepting a public firm bid—cash prices in the form of public firm bids are posted during harvest and for warehoused grain by merchants on a site basis. Growers can sell their parcel of grain immediately by accepting the price on offer via an online facility and then transfer the grain online to the buyer. The availability of this option depends on location and commodity.
- Placing an anonymous firm offer—growers can place a firm offer price on a parcel of grain anonymously and expose it to the entire market of buyers, who then bid on it anonymously using the Clear Grain Exchange, which is an independent online exchange. If the offer and bid match, the particulars of the transaction are sent to a secure settlement facility, although the title on the grain does not transfer from the grower until they receive funds from the buyer. The availability of this option depends on location and commodity. Anonymous firm offers can also be placed to buyers by an intermediary acting on behalf of the grower. If the grain sells, the buyer and seller are disclosed to each counterparty.

Counterparty risk

Most sales involve transferring the title on the grain prior to being paid. The risk of a counterparty defaulting when selling grain is very real and must be managed. Conducting business in a commercial and professional manner minimises this risk.

Principle I: Seller beware.

There is not much point selling for an extra $5/t if you don't get paid.

Counterparty risk management includes:

- Dealing only with known and trusted counterparties.
- Conducting a credit check (banks will do this) before dealing with a buyer they are unsure of.
- Selling only a small amount of grain to unknown counterparties.
- Considering credit insurance or a letter of credit from the buyer.
- Never delivering a second load of grain if payment has not been received for the first.
- Not parting with the title before payment, or requesting and receiving a cash deposit of part of the value ahead of delivery. Payment terms are negotiated at time of contracting. Alternatively, the Clear Grain Exchange provides secure settlement whereby the grower maintains title on the grain until they receive payment, and then title and payment are settled simultaneously.

Above all, act commercially to ensure the time invested in implementing a selling strategy is not wasted by poor management of counterparty risk. Achieving $5/t more on paper and not getting paid is a disastrous outcome.

Relative values

Grain-sales revenue is optimised when selling decisions are made in the context of the whole farming business. The aim is to sell each commodity when it is priced well, and to hold commodities that are not well priced at any given time. That is, give preference to the commodities with the highest relative value. This achieves price protection for the overall revenue of the farm business and enables more flexibility to a grower's selling program while achieving the business goal of reducing overall risk.

Principle J: Sell valued commodities, not undervalued commodities.

If one commodity is priced strongly relative to another, focus sales there. Don't sell the cheaper commodity for a discount.

For example, a farmer with wheat and barley to sell would sell the one that is getting good prices relative to the other, and hold the other for the meantime (Figure 18).

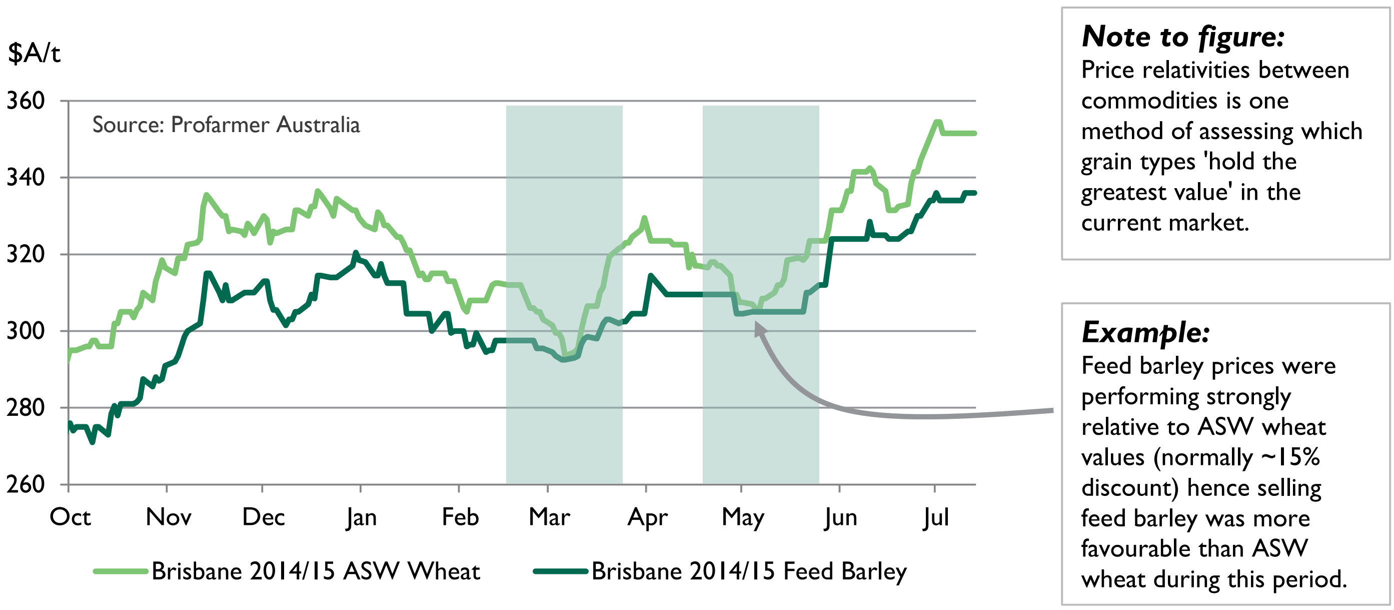


Figure 18: *Prices for Brisbane ASW wheat and feed barley are compared, and the barley held until it is favourable to sell it.*

Source: Profarmer Australia

If the decision has been made to sell wheat, CBOT wheat may be the better alternative if the futures market is showing better value than the cash market (Figure 19).

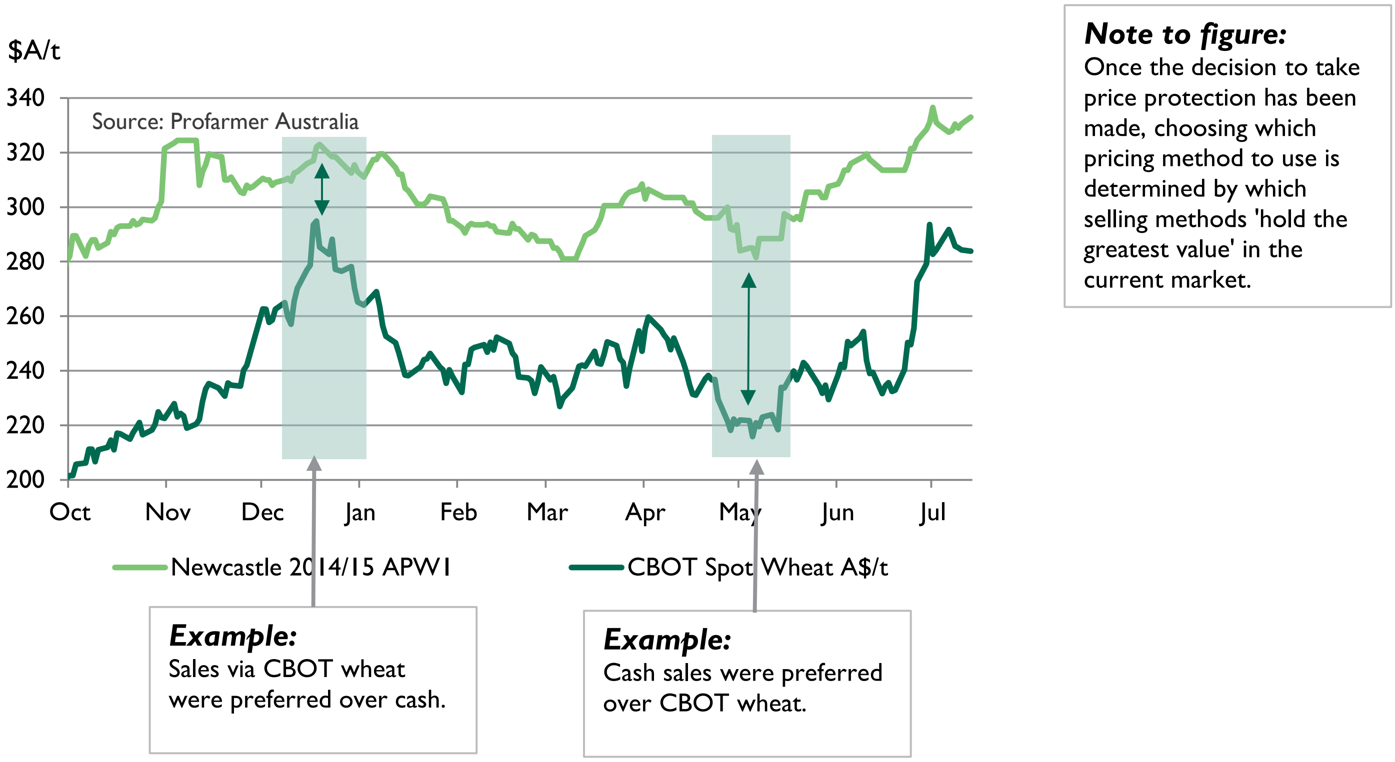


Figure 19: *Newcastle APWI and CBOT wheat prices (A$/t), showing when it is best to sell into each market.*

Source: Profarmer Australia

Principle K: Sell when there is buyer appetite.

When buyers are chasing grain, growers have more market power to demand the price they want.

Buyer appetite can be monitored by:

- The number of buyers at or near the best bid in a public bid line-up. If there are many buyers, it could indicate that buyer appetite is strong. However, if one buyer is offering $5/t above the next best bid, it may mean that cash prices are susceptible to falling $5/t as soon as that buyer satisfies their appetite.
- Monitoring actual trades against public indicative bids. When trades are occurring above indicative public bids it may indicate strong appetite from merchants and the ability for growers to offer their grain at price premiums to public bids. The chart below plots actual trade prices on the Clear Grain Exchange against the best public indicative bid on the day.

The selling strategy is converted to maximum business revenue by:

- Ensuring timely access to information, advice and trading facilities.
- Using different cash-market mechanisms when appropriate.
- Minimising counterparty risk by conducting effective due diligence.
- Understanding relative value and selling commodities when they are priced well.
- Thoughtful contract allocation.
- Reading market signals to extract value from the market or to prevent selling at a discount.

Contract allocation

Contract allocation means choosing which contracts to allocate your grain against come delivery time. Different contracts will have different characteristics (e.g. price, premiums-discounts, oil bonuses), and optimising your allocation reflects immediately on your bottom line.

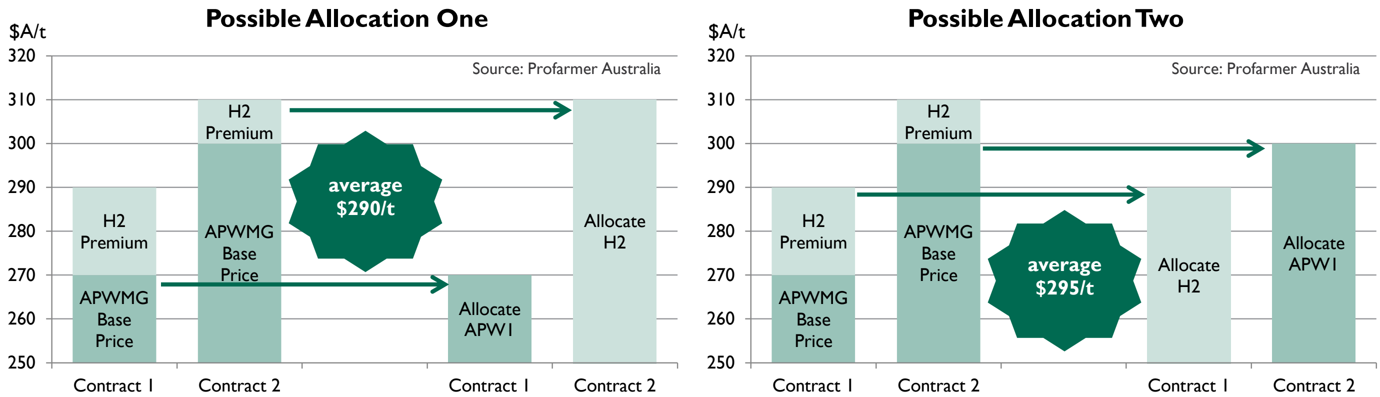
Principle L: Don't leave money on the table.

Contract allocation decisions don't take long, and can be worth thousands of dollars to your bottom line.

To achieve the best average price for their crop growers should:

- Allocate lower grades of grain to contracts with the lowest discounts.
- Allocate higher grades of grain to contracts with the highest premiums (Figure 20).

The grower may have several options. For example, Figure 20 shows that the only difference between achieving an average price of $290/t and $295/t is which contract each parcel is allocated to. Over an amount of 400 t, the difference in average price equates to nearly $2,000, which could be lost just in how parcels are allocated to contracts.



Note to figure:
In these two examples the only difference between acheiving an average price of $290/t and $295/t is which contracts each parcel was allocated to. Over 400/t that equates to $2,000 which could be lost just in how parcels are allocated to contracts.

Figure 20: *How parcels of the crop are allocated across contracts can make a substantial difference in income.*

Source: Profarmer Australia

Read market signals

The appetite of buyers to buy a particular commodity will differ over time depending on market circumstances. Ideally growers should aim to sell their commodity when buyer appetite is strong, and stand aside from the market when buyers are not very interested.

15.2 Southern chickpeas: market dynamics and execution

15.2.1 Price determinants for southern chickpeas

Australia is a relatively small player in terms of world pulse production, producing 1–2 million tonnes of pulses in any given year, compared to a global production of approximately 60 million tonnes. Chickpeas are the largest global pulse crop, with 11–12 million tonnes produced annually; field peas come in second with approximately 10 million tonnes. Australia's combined production of these crops is 1–1.3 million tonnes, or approximately 5% of global production.

Of the two major types of chickpeas grown in Australia, the desi chickpea is the predominant variety grown in NSW and Queensland, and the kabuli is more prominent in South Australia and Victoria.

Most of the desi chickpea crop is exported, and in terms of world trade, Australia is a major player. The major export markets for chickpeas are India and Pakistan, which between them import on average 1–1.5 million tonnes of chickpeas each year. In these markets, field peas can be used as a substitute to chickpeas. India imports 1.5–2.0 million tonnes of field peas each year.

Given this dynamic, Australian farm-gate prices are heavily influenced by global production volatility, international trade values into each of the major destinations, and price relativities between substitute products. For example, when India has a poor monsoon, Australian chickpea values tend to increase, as demand for imported product increases providing flow-on support to the Australian market. However, in years when Indian production is in surplus and import requirements are small, Australian product can become discounted, and Australia must seek other export destinations for local production. Because of Australia's involvement in international trade, it is important for growers to understand chickpea production in competitor nations (Figure 21) and the cropping calendar for chickpeas and field peas world wide (Figure 22).

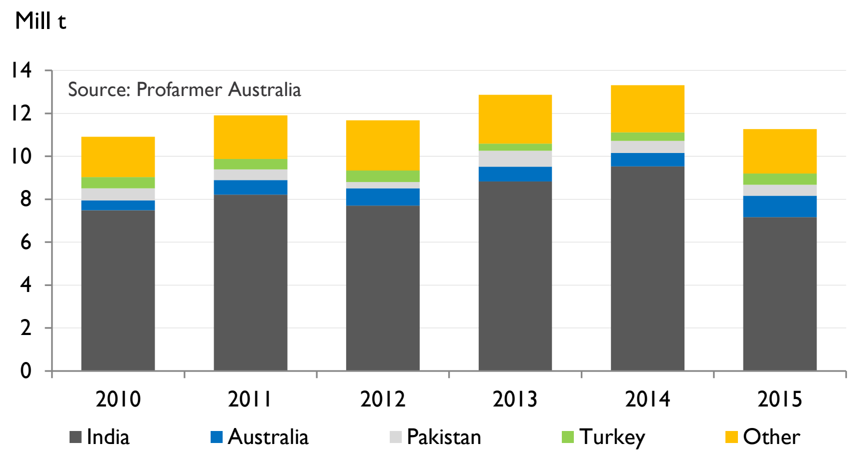


Figure 21: *World chickpea production*

Some of the global influences on Australian chick pea pricing are:

- Pulse production from the Indian domestic rabi cropping season (where harvest is April–May)—any negative influences will increase the need for imports of either chickpeas or field peas.
- The world price of field peas—field peas are purchased as a substitute pulse when the chickpea price is high.
- The timing of festivals in importing countries—Ramadan is the most important festival. It occurs in the ninth month of the Islamic calendar and goes for 29 days. Ramadan occurs around June then May for the next few years then will get closer to the end of the Australian harvest. This is favourable for supplying the Ramadan market post-harvest.

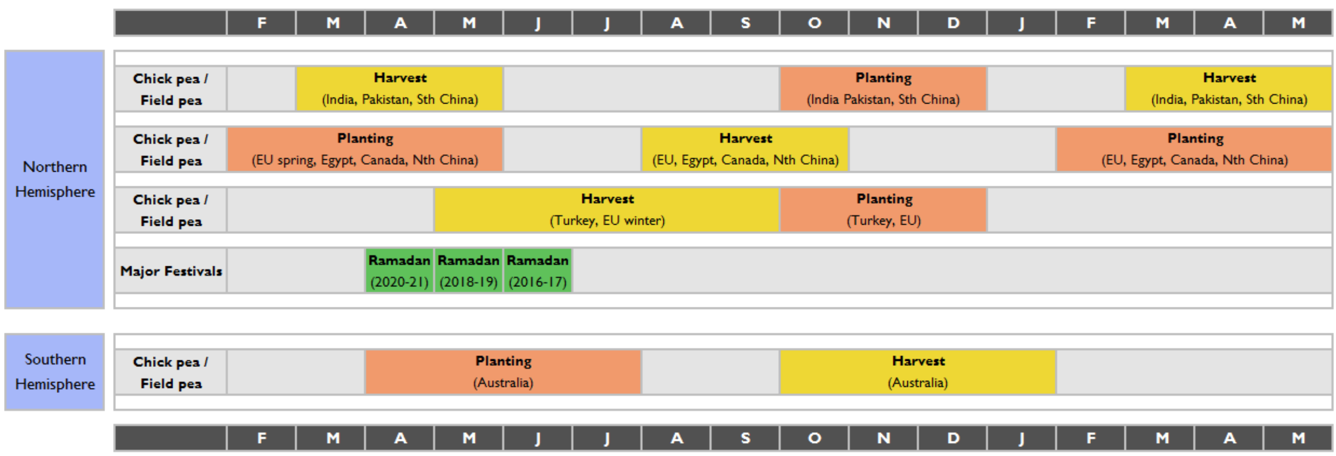


Figure 22: *Global field pea and chickpea crop calendar.*

The pace of Australian chickpea exports is typically strongest shortly after our harvest, as buyers seek to move crop ahead of the Indian harvest (Figure 23).

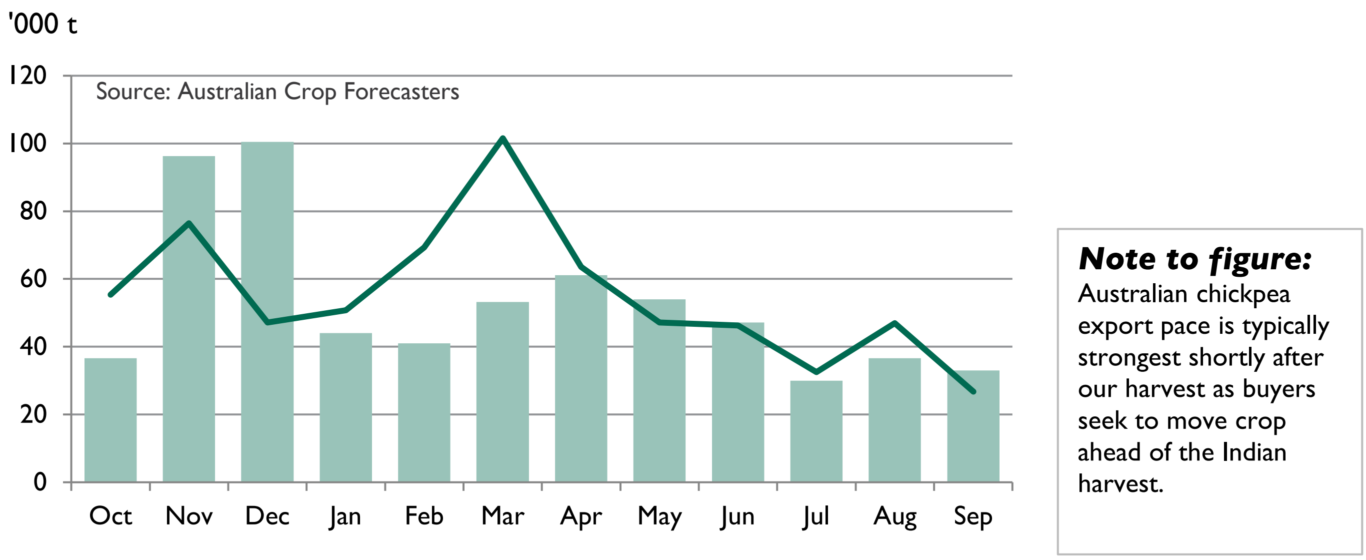


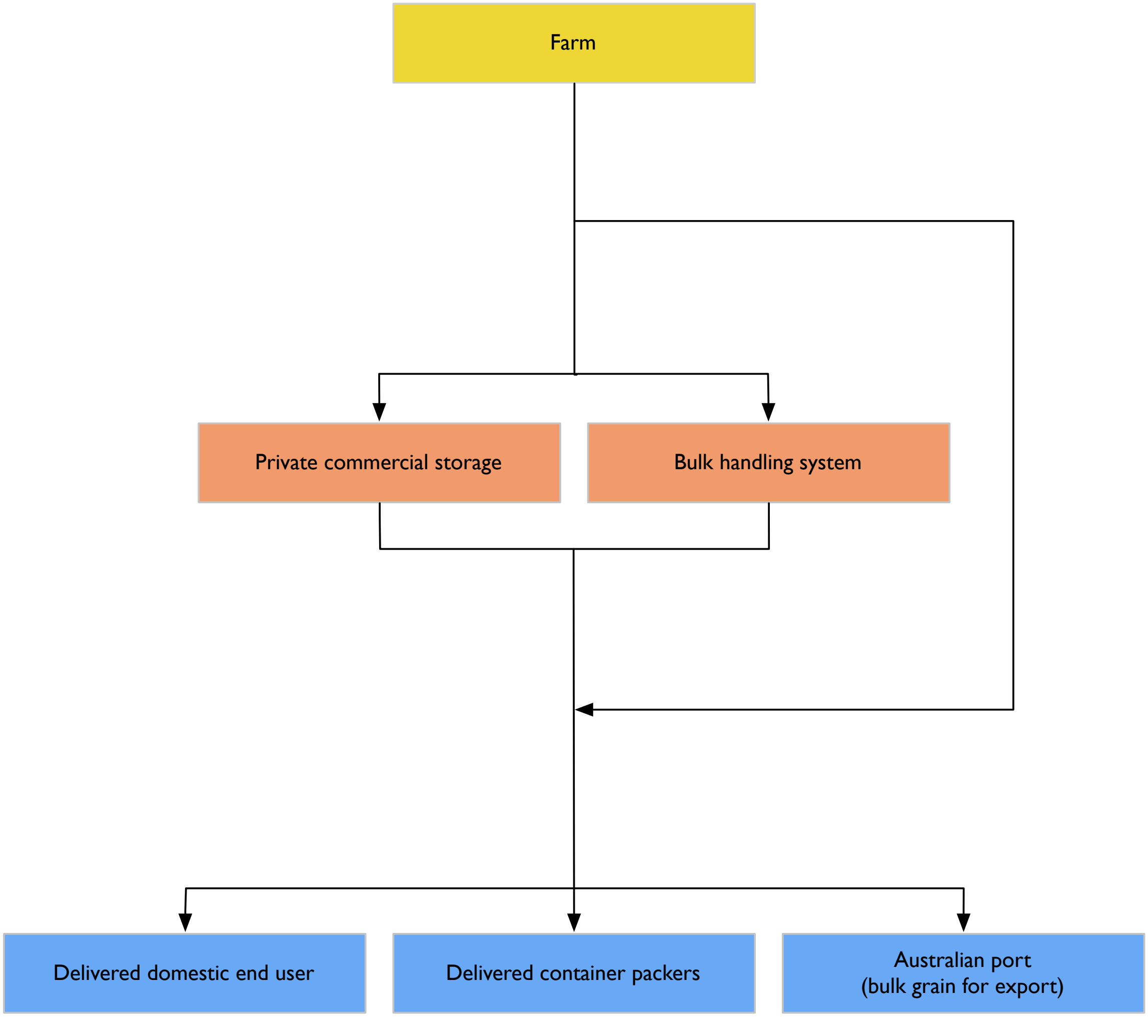
Figure 23: *Monthly export pace for chickpeas ('000 t).*

Source: Australian Crop Forecasters

15.2.2 Ensuring market access for chickpeas

The primary market for the chickpea crop is exports for human consumption. Most sales are exported in containers.

Chickpeas can also find homes as a source of protein in local stockfeed rations but at a lesser price. By and large, whether finding homes in export (generally via container) or domestic markets, private commercial storage and on-farm storage both provide efficient paths to market.



Note to figure:
Storage decisions should be determined by assessing market access. The large majority of northern canola is exported in bulk. Hence the bulk handling system should provide efficiencies to market. NSW also services a relatively large domestic market and private commercial and on-farm storage should provide a reasonable method to access these markets.

Profarmer AUSTRALIA

Figure 24: *Australian supply chain.*

15.2.3 Converting tonnes into cash for southern chickpeas

Given the volatile nature of chickpea pricing, setting a target price using the principles outlined minimises the risk of taking an unprofitable price or holding out for an unrealistically high price that may not eventuate.

The selling options for chickpeas are:

1. Store on farm then sell—this is the most common option. Chickpeas are relatively safe to store and require less maintenance than cereal grains. It is still important to monitor and maintain quality, as chick peas must meet strict quality specifications for export in order to avoid being discounted at the time of delivery. The grower must take into account cost of storage when setting target prices.
2. Cash sale at harvest—this is the least preferred option as buyer demand does not always coincide with harvest. Values can come under pressure at harvest time if a sudden increase in grower selling occurs in a small window, providing buyers with the confidence that they can meet their short- and medium-term commitments.

MORE INFORMATION

World pulse production calendar, in Pulses: Understanding global markets

Australian pulse traders

Understanding global markets: chickpeas

Chickpea marketing and standards

AEGIC, Australian pulses

Agriculture Victoria, Growing chickpea

3. Warehouse then sell—this option provides flexibility for sales if on-farm storage is not available. The grower must take into account warehousing costs as part of the cost of production when setting target prices. Warehousing is unlikely to be available to northern growers, as the major bulk handlers do not provide this option due to the low volume of production. The availability of this option from packers within the 'delivered' market will vary depending on the individual buyer.

As with all sales, a thorough understanding of counterparty risk and the terms of the contract of sale is essential. Counterparty risk considerations are especially important for pulse marketing, as there is a higher risk of contract default in international pulse markets than for canola or cereals. This is due to the markets they are traded into; the lack of appropriate price-risk tools (such as futures); and, often, the visual and subjective nature of quality determination. This can place extra risk on Australian-based traders endeavouring to find buyers for their product.

Current and past research

Project Summaries

As part of a continuous investment cycle each year the Grains Research and Development Corporation (GRDC) invests in several hundred research, development and extension and capacity building projects. To raise awareness of these investments the GRDC has made available summaries of these projects.

These project summaries have been compiled by GRDC's research partners with the aim of raising awareness of the research activities each project investment.

The GRDC's project summaries portfolio is dynamic: presenting information on current projects, projects that have concluded and new projects which have commenced. It is updated on a regular basis.

The search function allows project summaries to be searched by keywords, project title, project number, theme or by GRDC region (i.e. Northern, Southern or Western Region).

Where a project has been completed and a final report has been submitted and approved a link to a summary of the project's final report appears at the top of the page.

The link to Project Summaries is https://grdc.com.au/research/projects

Final Report Summaries

In the interests of raising awareness of GRDC's investments among growers, advisers and other stakeholders, the GRDC has available final reports summaries of projects.

These reports are written by GRDC research partners and are intended to communicate a useful summary as well as present findings of the research activities from each project investment.

The GRDC's project portfolio is dynamic with projects concluding on a regular basis.

In the final report summaries there is a search function that allows the summaries to be searched by keywords, project title, project number, theme or GRDC Regions. The advanced options also enables a report to be searched by recently added, most popular, map or just browse by agro-ecological zones.

The link to the Final Report Summaries is http://finalreports.grdc.com.au/final_reports.php

Online Farm Trials

The Online Farm Trials project brings national grains research data and information directly to the grower, agronomist, researcher and grain industry community through innovative online technology. Online Farm Trials is designed to provide growers with the information they need to improve the productivity and sustainability of their farming enterprises.

Using specifically developed research applications, users are able to search the Online Farm Trials database to find a wide range of individual trial reports, project summary reports and other relevant trial research documents produced and supplied by Online Farm Trials contributors.

The Online Farm Trials website collaborates closely with grower groups, regional farming networks, research organisations and industry to bring a wide range of crop research datasets and literature into a fully accessible and open online digital repository.

Individual trial reports can also be accessed in the trial project information via the Trial Explorer.

The link to the Online Farm Trials is http://www.farmtrials.com.au/

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