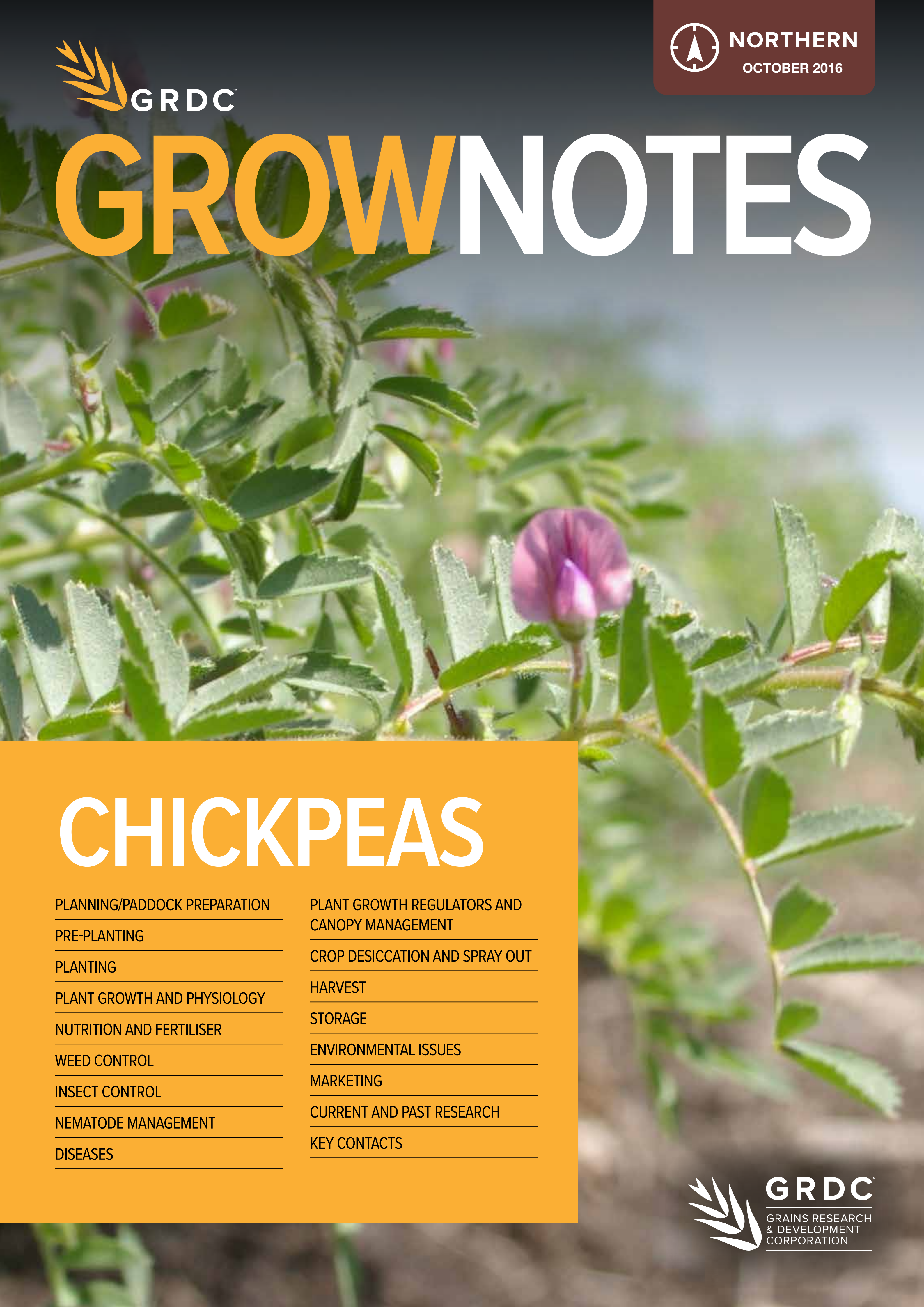

NORTHERN

OCTOBER 2016

GRDC

GROWNOTES

CHICKPEAS

GRDC

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- 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.
- Sow in an up and back row formation.
- 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. [1]

A.2 Crop overview

Chickpeas were first grown in Australia as a commercial crop in Goondiwindi, Queensland, during the early 1970s.

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.

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 (Figure 1) and are favoured in the Asian sub-continent.

Kabuli have large, rounder seeds, weighing about 400 mg. They are white–cream in colour and are almost exclusively used whole. They are preferred through the Mediterranean region.

The plant is erect and freestanding, ranging in height from 40-60 cm in the northern region, 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,

[1] Pulse Australia, Checklist for Northern Growers http://www.pulseaus.com.au/storage/app/media/crops/2010_NPB-Chickpea-checklist-north.pdf

and fine, frond-like 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. [2]

Figure 1: Desi chickpeas are split to obtain dhal.

More information

http://www.gograins.com.au/

http://www.grdc.com.au/Media-Centre/Ground-Cover/Ground-Cover-Issue-91-March-April-2011/Go-Grains

A.2.1 Nutritional Information

Chickpeas are a very good source of carbohydrates and proteins, which together constitute about 80% of the total dry seed weight. Starch, which is the principal carbohydrate component, varies in content from 41 to 50% and is lower in Desi varieties than in Kabuli varieties. Total seed carbohydrates vary from 52 to 71%. The crude protein content of chickpea varieties ranges from 16 to 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.

A.2.2 Pulses

Chickpeas are pulses, which by definition are annual legume crops that fix nitrogen from the atmosphere and produce high-protein grain for human consumption. [3]

Chickpeas are an annual leguminous crop, and the grain is used for human and animal consumption. 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. Pulses are the major source of protein in vegetarian diets. They have a

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

[3] 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

protein percentage of 20–25%, compared with wheat, which has half this and rice, with only one-third. [4]

The crop is generally sown in winter and harvested in late spring or summer. New South Wales is the state with the highest production, particularly northern NSW, followed by Queensland. Chickpeas are also grown in Western Australia, South Australia and Victoria.

Kabuli chickpeas are creamy-white and much larger than Desi chickpeas. 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.

The majority of Australian-produced chickpeas are exported, with India, Pakistan and Bangladesh taking nearly 80% of all exported chickpeas in the year ended October 2015. 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. [5]

A.2.3 Production

Table 1: Chickpea production 2016 for Desi and Kabuli chickpeas.

Desi chickpeas									
Region	Western	Southern				Northern			Australia Total
State	WA	SA	VIC	S/NSW	Subtotal	QLD	N/NSW	Subtotal	
2016 Production (t)	2,500	3,000	3,300	41,000	47,300	555,000	365,000	920,000	987,800
2016 Sown area (ha)	2,200	3,000	6,100	30,000	39,100	338,000	235,000	573,000	614,300
Variation from Dec 2015 (t)	-1,260	0	-1,600	0	-1,600	0	0	0	-2,860

Kabuli chickpeas									
Region	Western	Southern				Northern			Australia Total
State	WA	SA	VIC	S/NSW	Subtotal	QLD	N/NSW	Subtotal	
2015 Production (t)	700	8,000	2,000	3,100	13,100		29,400	29,400	43,200
2015 Sown area (ha)	500	13,900	6,800	2,800	23,500		23,000	23,000	47,000
Variation from Dec 2015 (t)	0	0	-500	0	-500		0	0	-500

(Source: Pulse Australia)

A.2.4 Quality attributes and end use

Australia is the world's number one chickpea exporter with 90% of Australian chickpeas exported and supplying more than a third of Desi chickpea traded internationally. 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

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

5 P Chudleigh (2012) An economic analysis of GRDC investment in the National Chickpea Breeding Program. GRDC Impact Assessment Report Series, December 2012, https://www.grdc.com.au/Research-and-Development/~/media/2FE8D5C5C0FE42B8BC7985647002FD70.pdf

- hydration and cooking characteristics of Desi and Kabuli chickpeas [6]

Chickpeas are prepared and eaten in a variety of ways (Figure 2). 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.

Figure 2: Chickpeas are exported for human consumption.

Chickpeas are a winter crop and, because they are leguminous, are valuable as an alternative crop in a cereal-based farming system. They are also an excellent break crop from diseases, weeds and pests. [7]

A.2.5 Role within northern region farming systems

Long-term tillage and rotation trials have been conducted by the Department of Agriculture, Fisheries and Forestry Queensland (DAF) and the New South Wales Department of Primary Industries (NSW DPI). These trials have shown that, through the adoption of best management practice (BMP) and breaking cereal monocultures by allocating 15–20% of winter cropping area to chickpeas, leading growers in Queensland and NSW have seen an average increase of 1 t/ha in yield and 1% in grain protein content in their following wheat crops.

Queensland chickpea production grew seven-fold between 1995 and 2005, and is continuing to increase. Agronomic guidelines for BMP are widely available through the websites and publications of the DAF, NSW DPI and Pulse Australia. Growers also have access to the network of chickpea agronomists accredited by Pulse Australia or have the opportunity to undertake the training themselves.

Chickpeas are recognised as a reliable, profitable winter crop with a vital role in farming systems and have great potential throughout the whole of the northern grains region. New chickpea varieties are released through Pulse Breeding Australia (PBA), a joint venture between Grains Research and Development Corporation (GRDC) and state departments of agriculture. [8]

6 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

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

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

A.2.6 GRDC's chickpea-breeding investment

A highly coordinated chickpea-breeding program commenced in Australia in 2005, but improvement via selection began in the 1970s. NSW DPI and DAFF have also collaborated on breeding chickpeas since 1983. Several improved varieties had been released in the period before the current investment commenced.

The principal outputs of GRDC chickpea-breeding investments have been improved varieties (Figure 3). Important traits from these improved varieties have been disease and pest resistance and traits that influence yield. 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.

Figure 3: The GRDC-funded chickpea-breeding program has resulted in improved varieties with better disease and pest resistance.

GRDC's investment in three projects (DAN00065, DAN00094, DAN00151) 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%. [9]

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 (DAF Qld)

9 P Chudleigh (2012) An economic analysis of GRDC investment in the National Chickpea Breeding Program. GRDC Impact Assessment Report Series, December 2012, https://www.grdc.com.au/Research-and-Development/~/media/2FE8D5C5C0FE42B8BC7985647002FD70.pdf

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)

More information

An Economic Analysis of GRDC Investment in the National Chickpea Breeding Program

Chickpea genome decoded, The Crawford Fund

www.grdc.com.au/Research-and-Development/Major-Initiatives/PBA

A.3 Keywords

Chickpeas, Desi, Kabuli, pulse, nitrogen fixation, rotation, breeding, northern farming systems.

SECTION 1

Planning/Paddock preparation

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). This helps to reduce the spread of the foliar and stubble-borne disease, Ascochyta blight. Avoid paddocks with high weed burdens, as chickpeas provide poor competition for weeds.

Prior to sowing, growers are advised to discuss with their agronomists their herbicide use over the previous 1–2 years and any potential residue problems. They should also understand crop management and harvest problems created by unlevelled paddocks and paddock obstacles.

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

- pH 5.2–8.0
- soil type—loams to self-mulching clays
- sodicity—avoid exchangeable sodium percentage (ESP) levels ≥3
- salinity, chloride—avoid electrical conductivity (EC) levels >1.5 dS/m
- potential waterlogging problems
- amount of stored soil moisture and received rainfall, noting their potential impact on herbicide residues [1]

Chickpeas established by direct drilling into standing cereal stubble reliably yield 10% higher than when using other planting techniques (Figure 1). [2]

1 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

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

More information

[NSW DPI Winter crop variety sowing guide]

Figure 1: Better chickpea yields are achieved by sowing into cereal stubble. (Photo: Gordon Cumming, Pulse Breeding Australia.)

1.1 Chickpea requirements

Chickpeas prefer well-drained loam to clay soils with a pH in the range 6–8. They will not grow in light acid soils. Areas prone to waterlogging should be avoided. This advice also applies to stony ground, because the plants need to be harvested close to the ground.

Chickpea are susceptible to hostile subsoils, with boron toxicity, sodicity and salinity causing patchiness in affected paddocks. Exchangeable aluminium tolerance in chickpea is nil. Tolerance to sodicity in the root-zone (to 90 cm) is <1% ESP on the surface and <3% ESP in subsoil (Table 1).

Broadleaf weeds and herbicide-resistant grasses can cause major problems, and a careful management strategy must be prepared well in advance. It may be possible to control the weeds in the year prior to cropping; however, paddocks with specific weeds that cannot be controlled by herbicides should be avoided.

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. [3]

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

Table 1: Pulse crop soil requirements

Crop	Soil type	Soil pH (CaCl2)	Exchangeable aluminium (%)	Drainage tolerance and rating (1–5)	Sodicity in root-zone (90 cm) exchangeable sodium percentage
Lupin, narrow leaf	Sandy loams	4.2–6.0	20% tolerant	Sensitive (2)	<1 surface <3 subsoil
Lupin, albus	Sandy loams, clay loams	4.6–7.0	Up to 8%	Very sensitive (1)	<1 surface <3 subsoil
Field pea	Sandy loams, clays	4.6–8.0	Up to 5–10%	Tolerant (3)	<1 surface <3 subsoil
Chickpea	Loams, self-mulching clay loams	5.2–8.0	Nil	Very sensitive (1)	<1 surface <3 subsoil
Faba bean	Loams, clay loams	5.4–8.0	Nil	Very tolerant (4)	<1 surface <3 subsoil
Canola	Loams, clay loams	4.8–8.0	0–5%	Tolerant (3)	<1 surface <3 subsoil
Lucerne	Loams, clay loams	5.0–8.0	Nil	Sensitive–tolerant (1–3)	<1 surface <3 subsoil

Source: G. Mullen (2004) Central NSW soils, NSW DPI.

More information

NSW DPI Paddock selection after drought

QDAF (2015), Planting chickpeas.

1.2 Paddock selection

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.

1.2.1 Avoid major variations in soil types

Crop maturity can be significantly affected by moisture supply during the growing season. Any major changes in soil type and moisture storage capacity across a paddock can lead to uneven crop maturity, delayed harvest, increased risk of weather damage, and/or high harvest losses due to cracking and splits. Uneven crop development also complicates timing of insecticide sprays, timing of desiccation, and management of Ascochyta blight.

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.

1.2.2 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.

1.2.3 Problematic paddocks

Stones and sticks are a concern in poorly or recently cleared country. 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.

1.2.4 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. [4]

1.3 Benefits of chickpeas as a rotation crop

1.3.1 Chickpeas preceding wheat

The benefits are:

- improved soil friability
- expanded weed-control options
- a break for diseases such as crown rot in wheat
- improved nitrogen (N) supply for cereal crops
- improved soil health

Risks:

- Poor weed competition. There is potential for build-up of Group A herbicide resistant wild oats, phalaris, and annual ryegrass, as there are few options registered for chickpeas. There is also potential for an increase in seed-bank population of problematic and hard-to-control weeds such as wireweed and climbing buckwheat.
- Nematodes are a major drawback to planting chickpeas before wheat. Recent field data show consistent differences in *Pratylenchus thornei* resistance between commercial chickpea varieties. Figure 2 shows a summary of performance of key chickpea varieties in eight trials sampled by Department of Agriculture, Fisheries and Forestry Queensland (DAFF), New South Wales Department of Primary Industries or Northern Grower Alliance (NGA).

More information

GRDC Update paper, 2013: Managing root lesion nematode: how important are crop and variety choice

A Verrell (2016), Integrated management of crown rot in a chickpea – wheat sequence

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

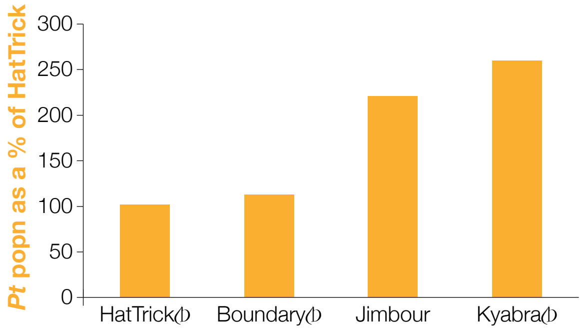

Figure 2: All varieties evaluated in 9 trials 2011-13 (DAFF, NSW DPI, NVT and NGA)

Chickpea is the preferred broadleaf rotation crop in the predominantly cereal farming systems of the northern region. The crop brings many benefits to the farming system and is currently the most adapted of all rotation crops to the climate, soils and the no-till farming systems of the north.

Chickpeas also provide flexibility:

- It is a profitable crop in its own right.
- Crop can be sown in wide rows (up to 100 cm) using a no-till system, which can offset the potential for small yield loss.
- Band spraying of wide rows reduces the amount of pesticide in the environment.
- Shielded spraying between wide rows allows rotation of chemistry for weed control.
- Ability to deep-sow provides the opportunity to plant on time most years.

More information

https://grdc.com.au/uploads/documents/Managing-N-for-Northern-Grains-Cropping.pdf

Pulse Australia (2016), Chickpea Production: Northern Region

NSW DPI (2015), Nitrogen benefits of chickpea and faba bean

Depending on the scale of the farm operation, other winter rotation crops and summer crops could be integrated into a rotation that contains chickpeas. Several long-term rotation trials have quantified the benefits of chickpeas to following cereal crops and to the overall farming system. There will always be variances between soil types, rainfall patterns and a range of other factors influencing the final yield outcome. 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 (see Table 2). [5]

Legumes must be well nodulated for maximum N fixation and soil N benefits. In most situations, growers will need to inoculate at sowing to ensure good levels of nodulation. If nodulation is adequate, legume N fixation is strongly and positively linked to productivity, and is suppressed by soil nitrate. Higher yields of legume crops also mean higher N and greater yield benefits for the following cereal crop. [6]

[5] M Lucy, D McCaffery, J Slatter (2006) Northern grain production—a farming systems approach. 2nd edn. Pulse Australia Ltd, http://www.pulseaus.com.au/storage/app/media/crops/2006_Pulses-northern-grain-farming-systems.pdf

[6] D Herridge (2013) Managing legume and fertiliser N for northern grains cropping. Revised version. GRDC 31 May 2013, http://www.grdc.com.au/GRDC-Booklet-ManagingFertiliserN

Table 2: Summary of a decade of experimental results from the northern grains belt showing the rotational benefits of chickpea on yield and grain protein levels of the following wheat crop, with and without fertiliser nitrogen (N)

Sites/rotations	Nil fertiliser N		Fertiliser N (75–150 kg N/ha)	
	Yield (t/ha)	Protein (%)	Yield (t/ha)	Protein (%)
New South Wales				
Chickpeas	1.9		1.9	
Wheat after wheat	2.1	11.2	2.7	13.2
Wheat after chickpeas	2.8	12.2	2.9	13.8
Queensland				
Chickpeas	1.5			
Wheat after wheat	2.2	10.3	2.8	13.8
Wheat after chickpeas	2.8	11.7	3.1	13.8

Source: Lucy et al. (2005).

Chickpeas provide many benefits in northern cropping rotations, including the ability to fix atmospheric nitrogen (N2), resulting in more soil N for following cereal crops. The amount of nitrogen fixed is determined by how well the pulse crop grows and the level of nitrate in the soil at planting. Soil nitrate suppresses nodulation and nitrogen fixation. Thus, high soil nitrate means low nitrogen fixation.

The nitrate-N benefit from chickpeas over a range of grain yields is shown in Table 3.

For chickpeas growing in the low nitrate soil, the nitrate-N benefit is consistently positive, ranging between 27 kg and 43 kg nitrate-N/ha over the range of yields (1.0–3.5 t/ha). In the moderate nitrate soil, the nitrate-N benefit of chickpeas essentially disappears. The simple message to take from this is that chickpeas should be grown in low nitrate soils so that they can fix large amounts of nitrogen, add to the soil's nitrogen fertility (balance) and, importantly for short-term productivity, increase the amount of nitrate-N in the root zone. [7]

Table 3: Nitrate-N benefit from chickpea over a range of grain yields.

		Low soil nitrate at sowing (50 kg N/ha)			Mod soil nitrate at sowing (100 kg N/ha)		
Grain yield (t/ha)	Shoot dry matter (t/ha)	N fixed (kg/ha)	N balance (kg/ha)#	Nitrate-N benefit (kg/ha)	N fixed (kg/ha)	N balance (kg/ha)#	Nitrate-N benefit (kg/ha)
1.0	2.7	37	1	35	22	-14	16
1.5	3.9	72	18	28	50	-5	3
2.0	5.1	110	40	27	80	9	-5
2.5	6.2	152	62	30	115	25	-9
3.0	7.2	195	88	35	150	43	-10
3.5	8.2	240	115	43	188	63	-8

(Source: NSW DPI)

1.3.2 The pulse effect on cereal yields

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.

[7] NSW DPI (2015), Nitrogen benefits of chickpea and faba bean. http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0009/572661/nitrogen-benefits-of-chickpea-and-faba-beans.pdf

More information

[GRDC Ground Cover, 2013: Trials measure chickpea/wheat rotation profit]

[GRDC Ground Cover, 2013: Trials explore chickpea/wheat rotations]

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, particularly crown rot, 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. 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.

Crown rot

Crown rot (caused by *Fusarium pseudograminearum*) is a major constraint to winter cereal production in Australia. The disease effectively blocks the base of infected tillers, preventing water movement from the roots through the stems and producing prematurely ripened heads (whiteheads) that contain no grain or lightweight shrivelled seed. Crown rot is a stubble-borne pathogen and survives as mycelium (cottony growth) inside cereal and grass weed residues.

The initial starting levels of crown rot inoculum and the season in which break crops are grown will influence their effectiveness. Hence, rotations to non-host winter pulses, oilseeds or summer crops are the most important component of an integrated disease management system.

The effectiveness of a break crop in reducing yield loss to crown rot is a function of both inoculum survival (decomposition) and water-use pattern of the break crop.

Chickpeas tend to use less water during the season than canola and generally do not root as deeply. Cereal crops following chickpea may experience reduced moisture stress through this water saving, thus reducing the development of whiteheads in infected tillers.

Yield response in a following cereal crop as a result of the benefit of reducing crown rot is a function of a break crop's effect on inoculum survival, and soil water and N.

Yield responses from NSW DPI experiments are summarised in Figure 3, where wheat yield following break crops is compared with other disease-management options of a long fallow, and early (December) or late (May) burning of wheat stubble.

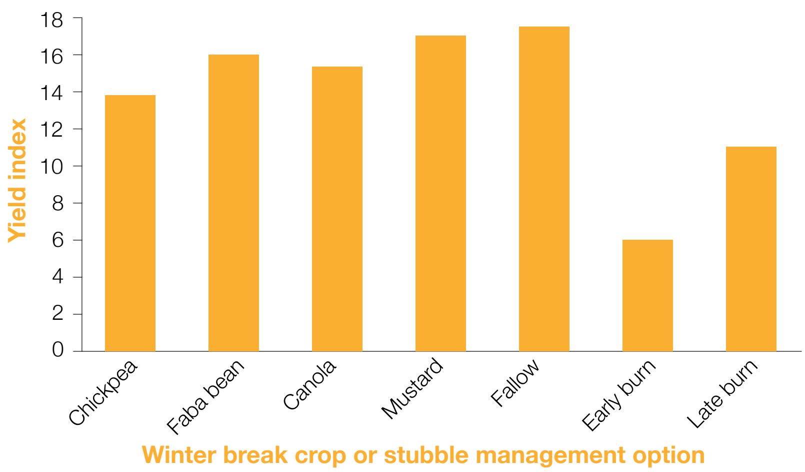

Figure 3: Yield index of wheat after various break crops of management options compared with continuous wheat. (Source: NSW DPI, 2006).

All winter and summer break-crop options provide a yield benefit in subsequent cereal crops as a direct result of reducing levels of crown rot. The selection of the appropriate break crop to suit individual situations will have the most economic benefit.

Quantifying yield increases after break crops

Yields of wheat grown after a broadleaf break crop generally exceed those of wheat grown after wheat or other cereals. The presumed reasons for the yield benefit vary between break crops. They include reduced root and foliar disease, increased supply of soil water and mineral N, reduced assimilate loss to mycorrhizae, and, after legumes, growth stimulation following hydrogen gas release.

Data used to evaluate the reasons for yield increase suggest that control of soil-borne disease, and residual N, after legumes are the largest benefits of break crops. [8]

1.4 Which rotation is best?

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.

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 3 years. 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.

To minimise foliar diseases, it is important to recognise that a distance of at least 500 m is needed between the planned pulse crop and the stubble of that pulse (or others) from the previous year.

1.4.1 Nitrogen fixation

A pulse crop does not necessarily add large quantities of N to the soil. 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. Thus, high soil nitrate means low N fixation (Figure 4).

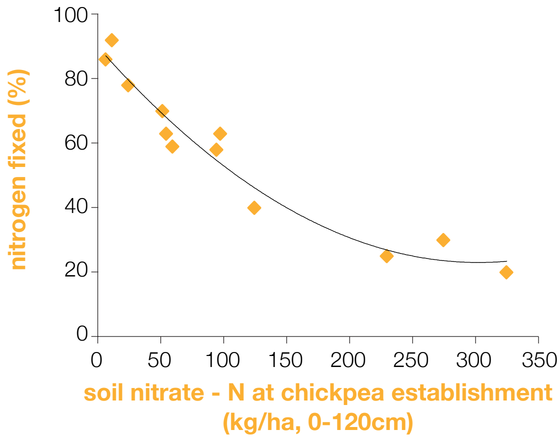

Figure 4: Effect of soil nitrate nitrogen on nitrogen fixation by chickpeas. (Source: J.A. Doughton et al 1993).

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

1.4.2 Nitrate-N benefit for following cereals

The nitrate-N benefit from chickpea over a range of grain yields has been calculated from trials in northern Australia (Herridge *et al*. 2003) and is 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.

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).

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

1.4.3 Availability of nitrogen

One of the tangible benefits of a pulse crop is the rapid breakdown of the N-rich organic matter remaining after the crop has been grown.

Most of this organic N is readily available to the following crop and is one of the reasons why cereal yields are often high following a pulse crop.

1.4.4 Weeds

A range of effective herbicides is available to control grasses in pulse crops. It is now possible to control wild oats and ryegrass.

However, control of herbicide-resistant weeds such as wild oats and annual ryegrass is now a major issue, and appropriate planning and management are needed.

Some of the weeds are not effectively controlled in the following cereal crop, but by controlling them in the pulse phase of a rotation and preventing them from setting seed, they can be virtually eliminated from the following cereal crop.

Pulses, particularly chickpeas, are poor weed competitors. Unless attention is paid to effective control measures, weeds are likely to build up, reducing yields and seriously affecting future crops.

Crop rotation is a key strategy in managing herbicide resistance, allowing for the use of herbicides with different modes of action from those used in cereal crops.

Many of the broadleaf weeds can be difficult to control and can cause substantial yield loss and grain contamination. Good knowledge of likely weed problems is needed, because most broadleaf herbicides are applied before sowing or before crop emergence. [9]

1.5 Disadvantages of chickpeas as a rotation crop

Chickpeas are also susceptible to Ascochyta blight, and as a high priority, new chickpea crops should be grown at least 500 m from chickpea stubble to avoid infection. [10]

Growers should aim for a break of at least 4 years between chickpea crops in each paddock. Chickpeas are intolerant to root-lesion nematodes, and some chickpea varieties lose up to 20% yield when nematode populations are high. [11]

Where profile N is high there may be little additional N fixed by chickpeas.

Chickpeas are poor competitors with weeds. [12]

Chickpeas are also susceptible to Phytophthora root rot, so avoid sowing chickpeas into paddocks with a history of any level of the disease, paddocks with a history of lucerne or medics, and paddocks prone to waterlogging and/or flooding.

More information

[Australian Pesticides and Veterinary Medicines Authority (APVMA)]

[GRDC Update Paper, 2014: Weeds and resistance considerations for awnless barnyard grass, chloris and fleabane (NGA)]

1.6 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, for example, double-cropped from sorghum or cotton, or areas with a sequence of clean winter fallows.

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.

More information

[GRDC Fact Sheet, 2014: Summer fallow spraying]

[C McMaster, N Graham, J Kirkegaard, J Hunt, I Menz (2015). "Buying a spring" – the water and nitrogen cost of poor fallow weed control.]

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 northern 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. [13]

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

10 W Hawthorne, J Davidson, L McMurray (2012) Chickpea disease management strategy. Pulse Australia Southern Pulse Bulletin #8, http://pulseaus.com.au/storage/app/media/crops/2012_SPB-Chickpea-disease-management.pdf

11 DAFF (2009) Management of root-lesion nematodes in the northern grain region. Department of Agriculture Fisheries and Forestry, http://www.daf.qld.gov.au/__data/assets/pdf_file/0010/58870/Root-Lesion-Nematode-Brochure.pdf

12 W Hawthorne, J Davidson, L McMurray (2012) Chickpea disease management strategy. Pulse Australia Southern Pulse Bulletin #8, http://pulseaus.com.au/storage/app/media/crops/2012_SPB-Chickpea-disease-management.pdf

13 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

The NGA is trialing methods to control summer grasses. Key findings so far include:

1. Glyphosate-resistant and -tolerant weeds are a major threat to our reduced-tillage cropping systems.
2. Although residual herbicides will limit recropping options and will not provide complete control, they are key to successful fallow management.
3. 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.
4. Other weed management tactics can be incorporated (e.g. crop competition, to assist herbicide control).
5. Cultivation may need to be considered as a salvage option to avoid seed-bank salvage.

More information

GRDC Fact Sheet,2012: Effective Double Knock Herbicide Applications Northern Region

Australian Herbicide Resistance Initiative (2015). Knock knock.

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. [14]

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 (e.g. awnless barnyard grass, ABYG) or when growing conditions are very favourable. Longer intervals are needed for weeds where translocation appears slower (e.g. fleabane, feathertop Rhodes grass and windmill grass).

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 rarely effective for salvage weed-control situations unless environmental conditions are exceptionally favourable.

Important weeds in northern cropping systems

Weed management, particularly in reduced tillage fallows, has become an increasingly complex and expensive part of cropping in the northern grains region. Heavy reliance on glyphosate has selected for species that were naturally more glyphosate-tolerant or has selected for glyphosate-resistant populations. The four key weeds that are causing major cropping issues are:

1. Awnless barnyard grass (*Echinochloa colona*)
2. Flaxleaf fleabane (*Conyza bonariensis*)
3. Feathertop Rhodes grass (FTR) (*Chloris virgata*)
4. Windmill grass (*Chloris truncata*)

14 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

Awnless barnyard grass

Figure 5: Awnless barnyard grass. (Photo: Rachel Bowman.)

Awnless barnyard grass (Figure 5) has been a major summer grass problem for many years. It is a difficult weed to manage for at least three main reasons:

1. It has multiple emergence flushes (cohorts) each season.
2. It is easily moisture-stressed, leading to inconsistent knockdown control.
3. Glyphosate-resistant populations are increasingly being found.

Key points

- Glyphosate resistance is widespread. Tactics against this weed must change from glyphosate alone.
- Utilise residual chemistry wherever possible and aim to control 'escapes' with camera spray technology, such as Weedseeker or WeedIt.
- Try to ensure that a double-knock of glyphosate followed by paraquat is used on one of the larger early-summer flushes of ABYG.
- Restrict Group A herbicides to management of ABYG in-crop and aim for strong crop competition.

Resistance levels

Prior to summer 2011–12, there were 21 cases of glyphosate-resistant ABYG. Collaborative surveys were conducted by NSW DPI, DAFF and NGA in summer 2011–12 with a targeted follow-up in 2012–13. Agronomists from the Liverpool Plains to the Darling Downs and west to areas including Mungindi collected ABYG samples, which were tested at the Tamworth Agricultural Institute with Glyphosate CT at 1.6 L/ha (a.i. 450 g/L) at a mid-tillering growth stage. Total application volume was 100 L/ha.

The main finding from this survey work was that the number of 'confirmed' glyphosate-resistant ABYG populations had nearly trebled. Selected populations were also evaluated in a separate glyphosate rate-response trial. The experiment showed that some of these populations were suppressed only when sprayed with 12.8 L/ha.

Growers can no longer rely on glyphosate alone for ABYG control.

Residual herbicides (fallow and in-crop)

A range of active ingredients is registered in either summer crops, e.g. metolachlor (e.g. Dual Gold®) and atrazine, or fallow (e.g. imazapic, e.g. Flame®), and these provide useful management of ABYG. The new fallow registration of isoxaflutole (Balance®) can provide useful suppression of ABYG but has stronger activity against other problem weed species. Few (if any) residuals give consistent, complete control. However, they are important tools that need to be considered to reduce the weed population exposed to knockdown herbicides, as well as to alternate the herbicide chemistry being employed. Use of residuals together with camera spray technology (for escapes) can be a very effective strategy in fallow.

Double-knock control

This approach uses two different tactics applied sequentially. In reduced-tillage situations, it is frequently glyphosate first followed by a paraquat-based spray as the second application or 'knock'. Trials to date have shown that glyphosate followed by paraquat has given effective control even on glyphosate-resistant ABYG. Note that the most effective results will be achieved from paraquat-based sprays by using higher total application volumes (100 L/ha) and finer spray quality and by targeting seedling weeds.

Several Group A herbicides (e.g. Verdict® and Select®) are effective on ABYG but should be used in registered summer crops (e.g. mungbeans). Even on glyphosate-resistant ABYG, a glyphosate followed by paraquat double-knock is an effective tool. Note that Group A herbicides appear more sensitive to ABYG moisture stress. Application on larger, mature weeds can result in very poor efficacy.

Timing of the paraquat application for ABYG control has generally proven flexible. The most consistent control is obtained from a delay of about 3–5 days, when lower rates of paraquat can also be used. Longer delays may be warranted when ABYG is still emerging at the first application timing; shorter intervals are generally required when weed size is larger or moisture stress conditions are expected. High levels of control can still be obtained with larger weeds but paraquat rates will need to be increased to 2.0 or 2.4 L/ha.

Flaxleaf fleabane

Figure 6: Flaxleaf fleabane. (Photo: DAFF, Qld.)

There are three main species of fleabane in Australia: *Conyza bonariensis* (flaxleaf fleabane, Figure 6), *C. canadensis* (Canadian fleabane) and *C. albida* (tall fleabane).

There are two varieties of *C. canadensis*: var. *canadensis* and var. *pusilla*. Of the three species, flaxleaf fleabane is the most common across Australia. [15]

For more than a decade, flaxleaf fleabane (*C. bonariensis*) has been the major weed-management issue in the northern cropping region, particularly in reduced-tillage systems. Fleabane is a wind-borne, surface-germinating weed that thrives in situations of low competition. Germination flushes typically occur in autumn and spring when levels of surface soil moisture stay high for a few days. However, emergence can occur at nearly all times of the year.

An important issue with fleabane is that knockdown control of large plants in the summer fallow is variable and can be expensive due to reduced control rates.

Key points

- Utilise residual chemistry wherever possible and aim to control 'escapes' with camera spray technology.
- This weed thrives in situations of low competition; avoid wide row cropping unless effective residual herbicides are included.
- 2,4-D is a crucial tool for consistent double-knock control.
- Successful growers have increased their focus on fleabane management in winter (crop or fallow) to avoid expensive and variable salvage control in the summer.

More information

Australian Glyphosate Sustainability Working Group (AGSWG): Australian glyphosate resistance register

Resistance levels

Glyphosate resistance has been confirmed in fleabane. There is great variability in the response of fleabane to glyphosate, with many samples from non-cropping areas still well controlled by glyphosate, whereas fleabane from reduced-tillage cropping situations shows increased levels of resistance. The most recent survey has focused on non-cropping situations, with a large number of resistant populations found on roadsides and railway lines where glyphosate alone has been the principal weed management tool employed.

Residual herbicides (fallow and in-crop)

One of the most effective strategies to manage fleabane is the use of residual herbicides during fallow or in-crop. Trials have consistently shown good efficacy from a range of residual herbicides commonly used in sorghum, cotton, chickpea and winter cereals. There are now at least two registrations for residual fleabane management in fallow.

Additional product registrations for in-crop knockdown and residual herbicide use, particularly in winter cereals, are still being sought. A range of commonly used winter cereal herbicides exists with useful knockdown and residual fleabane activity. Trials to date have indicated that increasing water volumes from 50 to 100 L/ha may help the consistency of residual control, with application timing to ensure good herbicide/soil contact also important.

Knockdown herbicides (fallow and in-crop)

Group I herbicides have been the major products for fallow management of fleabane, with 2,4-D amine the most consistent herbicide evaluated. Despite glyphosate alone generally giving poor control of fleabane, trials have consistently shown a benefit from tank mixing 2,4-D amine and glyphosate in the first application. Amicide® Advance at 0.65–1.1 L/ha mixed with Roundup® Attack at a minimum of 1.15 L/ha and then followed by Nuquat® at 1.6–2.0 L/ha is a registered option for fleabane knockdown in fallow. Sharpen is a product with Group G mode of action. It is registered for fallow control when mixed with Roundup Attack at a minimum of 1.15 L/ha but only on fleabane up to a maximum of six leaves. There are no registered knockdown options in chickpeas.

More information

For more information on label rates, visit: www.apvma.gov.au

[15] M Widderick, H Wu. Fleabane. Department of Agriculture and Food Western Australia, http://www.agric.wa.gov.au/objtwr/imported_assets/content/pw/weed/major/fleabane.pdf

Double-knock control

The most consistent and effective double-knock control of fleabane has included 2,4-D in the first application followed by paraquat as the second. Glyphosate alone followed by paraquat will result in high levels of leaf desiccation but plants nearly always recover.

Timing of the second application in fleabane is generally aimed at about 7–14 days after the first application. However, the interval to the second knock appears quite flexible. Increased efficacy is obtained when fleabane is actively growing or if rosette stages can be targeted. Although complete control can be obtained in some situations (e.g. summer 2012–13) control levels will frequently reach only about 70–80%, particularly when targeting large, flowering fleabane under moisture-stressed conditions. The high cost of fallow double-knock approaches and inconsistency in control level of large, mature plants are good reasons to focus on proactive fleabane management at other growth stages.

More information

http://www.qaafi.uq.edu.au/content/Documents/weeds/IWM-Fleabane-guide.pdf

Feathertop Rhodes grass

Figure 7: Feathertop Rhodes grass. (Photo: Rachel Bowman)

Feathertop Rhodes grass (Figure 7) has emerged as an important weed-management issue in southern Queensland and northern NSW since about 2008. This is another small-seeded weed species that germinates on, or close to, the soil surface. It has rapid early growth rates and can become moisture stressed quickly. Although FTR is well established in central Queensland, it remains largely an 'emerging' threat further south. Patches should be aggressively treated to avoid whole-of-paddock blow-outs.

Key points

- Glyphosate alone or glyphosate followed by paraquat has generally poor efficacy.
- Utilise residual chemistry wherever possible and aim to control 'escapes' with camera spray technology.
- A double-knock of Verdict followed by paraquat can be used in Queensland prior to planting mungbeans where large spring flushes of FTR occur.
- Treat patches aggressively, even with cultivation, to avoid paddock blow-outs.

Residual herbicides (fallow and in-crop)

This weed is generally poorly controlled by glyphosate alone even when sprayed under favourable conditions at the seedling stage. Trials have shown that residual herbicides generally provide the most effective control, a similar pattern to that seen with fleabane. Currently registered residual herbicides are being screened and offer promise in both fallow and in-crop situations. The only product currently registered for FTR control is Balance (isoxaflutole) at 100 g/ha for fallow use.

Double-knock control

Whereas a glyphosate followed by paraquat double-knock is an effective strategy on

ABYG, the same approach is variable and generally disappointing for FTR management. By contrast, a small number of Group A herbicides (all members of the 'fop' class) can be effective against FTR but need to be managed within a number of constraints:

- Although they can provide high levels of efficacy on fresh and seedling FTR, they need to be followed by a paraquat double-knock to get consistent high levels of final control.
- Group A herbicides have a high risk of resistance selection, again requiring follow-up with paraquat.
- Many Group A herbicides have plant-back restrictions to cereal crops.
- Group A herbicides generally have a narrower range of weed growth stages for successful use than herbicides such as glyphosate (i.e. Group A herbicides will generally give unsatisfactory results on flowering and/or moisture-stressed FTR).
- Not all Group A herbicides are effective on FTR.

For information on a permit (PER12941) issued for Queensland only for the control of FTR in summer-fallow situations prior to planting mungbeans, see www.apvma.gov.au.

Timing of the second application for FTR is still being refined, but application at about 7–14 days generally provides the most consistent control. Application of paraquat at shorter intervals can be successful, when the Group A herbicide is translocated rapidly through the plant, but has resulted in more variable control in field trials. Good control can often be obtained up to 21 days after the initial application.

Windmill grass

Figure 8: Windmill grass. (Photo: Maurie Street)

While FTR has been a grass weed threat coming from Queensland and heading south, windmill grass (Figure 8) is more of a problem in central NSW and is spreading north. Windmill grass is a perennial, native species found throughout northern NSW and southern Queensland. The main cropping threat appears to be from the selection of glyphosate-resistant populations, with control of the tussock stage providing most challenges to management.

Key points

- Glyphosate alone or glyphosate followed by paraquat has generally poor efficacy.
- Preliminary data suggest that residual chemistry may provide some benefit.
- A double-knock of quizalofop-p-ethyl (e.g. Targa) followed by paraquat can be used in NSW.

Resistance levels

Glyphosate resistance has been confirmed in windmill grass, with three documented cases in NSW, all located west of Dubbo. Glyphosate-resistant populations of windmill grass in other states have all been collected from roadsides, but in central-western NSW, two were from fallow paddock situations.

Residual herbicides (fallow and in-crop)

Preliminary trials have shown a range of residual herbicides with useful levels of efficacy against windmill grass. These herbicides have potential for both fallow and in-crop situations. Currently, no products are registered for residual control of windmill grass.

Double-knock control

Similar to FTR, a double-knock of a Group A herbicide followed by paraquat has provided clear benefits compared with the disappointing results usually achieved by glyphosate followed by paraquat. Constraints apply to double-knock for windmill grass control similar to those for FTR.

More information

http://www.grdc.com.au/Media-Centre/Ground-Cover-Supplements/GCS102/Herbicide-resistance-in-summer-grasses

http://www.nga.org.au/module/documents/download/225

For information on a permit for NSW only for the control of windmill grass in summer fallow situations, visit www.apvma.gov.au.

Timing of the second application for windmill grass is still being refined, but application at about 7–14 days generally provides the most consistent control. Application of paraquat at shorter intervals can be successful, when the Group A herbicide is translocated rapidly through the plant, but has resulted in more variable control in field trials and has been clearly antagonistic when the interval is 1 day or less. Good control can often be obtained up to 21 days after the initial application. [16]

1.7 Fallow chemical plant-back periods

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)). This is shown in Table 4 where known. 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. [17]

16 R Daniel (2013) Weeds and resistance—considerations for awnless barnyard grass, *Chloris* spp. and fleabane management. Northern Grower Alliance, http://www.nga.org.au/module/documents/download/225

17 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

Table 5: Residual persistence of common pre-emergent herbicides, and noted residual persistence in broadacre trials and paddock experiences [18]

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 long-lasting 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
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

Sources: CDS Tomlinson (Ed.) (2009) The pesticide manual. 15th edn. British Crop Protection Council, Farnham, UK. Extoxnet: http://extoxnet.orst.edu/. California Dept Pesticide Regulation Environmental Fate Reviews, www.cdpr.ca.gov/

More information

www.apvma.gov.au

http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0003/431247/Using-pre-emergent-herbicides-in-conservation-farming-systems.pdf

Download the NSW DPI publication 'Weed control in winter crops' at: http://www.dpi.nsw.gov.au/agriculture/broadacre/guides/weed-control-winter-crops

1.7.1 Herbicide residues in soil

Residues from herbicides used in the current or previous crop could impact on subsequent crop choice in rotations. 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. 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. [19]

18 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

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

1.8 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 (Figure 9).

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 6 and Table 7 below to assist in testing and diagnosis.

Figure 9: Aerial shot of chickpea crops near Garah in 2012 showing wide-scale crop loss due to sodic/saline conditions.

Table 6: 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 7: 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			High EC >0.3 dS/m in top 10 cm		
Low EC <0.7 dS/m in subsoil			High EC >0.7 dS/m in subsoil		
Plant growth is not affected by salinity:			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.

More information

http://www.grdc.com.au/uploads/documents/dnr00004.pdf

1.8.1 Chemical constraints

Soil pH can affect plant growth and nutrient availability.

Acid 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 CaCl2) or 6.3 (in water). Monitor changes in soil pH by regular soil testing. If severe acidity is allowed to develop, then 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 have a high proportion of material with 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 marginal.

Low soil pH often leads to poor or ineffective nodulation in pulses because acid soil conditions affect rhizobial initial numbers and multiplication. Field peas, faba beans, lentil and chickpea 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 soil conditions.

Alkaline soils

In alkaline soils, the abundance of carbonates and bicarbonates can reduce crop growth and induce nutrient deficiencies. 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.

1.8.2 Sodic soils

Chickpeas are classified among the most sensitive of all field crops to sodic soil conditions (Table 8).

Soils high in sodium are structurally unstable, with clay particles dispersing when wet. This subsequently blocks soil pores, 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 also restrict seedling emergence.

Some indicators of surface sodicity include:

- soils prone to crusting and sealing up
- ongoing problems with poor plant establishment
- presence of scalded areas in adjoining pasture

Exchangeable sodium percentage is the measure for sodicity:

- 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 10 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 from depth.

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.

Table 8: Relative tolerance of crops to sodicity (high exchangeable sodium percentage)

Tolerant	Semi-tolerant	Sensitive
Rice	Barley	Maize
	Wheat	Cowpeas
	Cotton	Peanuts
	Sorghum	Mungbeans
		Lentils
		Sunflower
		Guar
		Chickpea

Source: Abrol (1973).

Bob Brinsmead (formerly Qld Department of Primary Industries and Fisheries) confirmed soil sodicity as the cause of plant death and low yields on certain brigalow soils in the Billa Billa and Talwood areas in 2000.

Crops were very patchy, with considerable stunting and eventual plant death under the very dry seasonal conditions. Areas that were less affected in the paddocks were yielding about 1 t/ha. Dieback appeared to be associated with the pattern of gilgai throughout the affected paddocks.

While there is no clear-cut association between topography and vegetation type with the occurrence of sodicity, the problem is more likely to occur in:

- brigalow and brigalow–belah land systems
- duplex red-brown earths
- poplar box on texture contrast soils
- ironbark/bulloak land systems

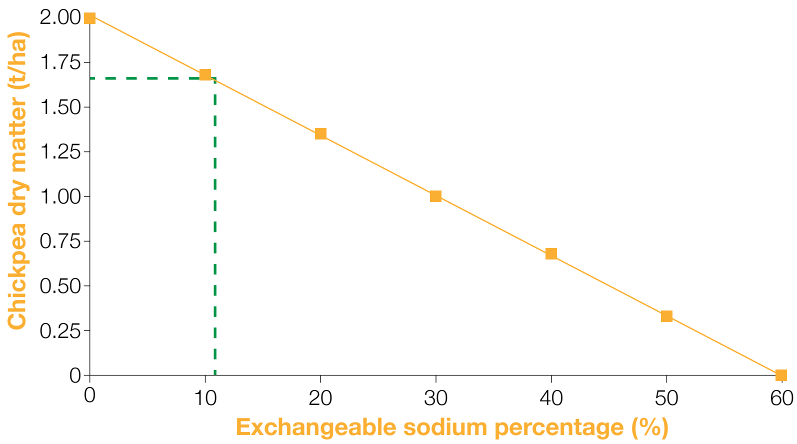


Figure 10: Impact of sodicity on chickpea dry matter production.

1.8.3 Salinity

Salinity is the presence of dissolved salts in soil or water. It causes iron toxicity in plants and impedes their ability to absorb water (see Figures 11 and 12 for typical salt effects).

Figure 11: Salt effects as seen on a soil of EC 11 ds/m.

Figure 12: Typical salt effects on chickpea leaves.

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 yields.

As with sodicity, saline soil deeper than 90 cm down the profile is unlikely to impact on chickpea yields; the closer the surface, the greater the detrimental effect.

All current varieties of chickpea are considered highly sensitive to salinity. Levels of EC >1.5 dS/m will cause a yield reduction in chickpea, whereas wheat will tolerate EC up to 6 ds/m with no yield reduction (Table 9, Table 10).

Table 9: Crop tolerances to salinity (EC, mmhos/cm = dS/m = mS/cm)

Crop	Expected yield reduction:				
	0%	10%	25%	50%	100%
Barley	8.0	10.0	13.0	18.0	56.0
Cotton	7.7	9.6	13.0	1.70	54.0
Wheat	6.0	7.4	9.5	13.0	40.0
Sorghum	4.0	5.1	7.2	11.0	36.0
Peanut	3.2	3.5	4.1	4.9	13.0
Maize	1.7	2.5	3.8	5.9	20.0
Faba bean	1.6	2.6	4.2	6.8	24.0
Chickpea	*1.3*	*2.0*	*3.1*	*4.9*	*8.0*
Beans	1.0	1.5	2.3	3.6	13

Adapted from: Mass and Hoffman (1977) and Abrol (1973).

Table 10: Relative tolerance of plants to soil salinity, determined as electrical conductivity (EC) level at which yield is reduced by 50%

Note that actual numerical values of EC depend on soil texture and should be taken as a guide only. Salt tolerance is further reduced in seedlings

Soil salinity for 50% yield reduction (EC, dS/m)	Crop, pasture	Rating
12.0	Puccinella	Highly tolerant
7.0	Salt bush	
4.8	Tall wheat grass	
4.5	Barley	
4.1	Canola	Tolerant
4.0	Cereal rye	
3.25	Bread wheat	
3.1	Triticale	
2.6	Sorghum	
2.5	Safflower	Moderately tolerant
2.25	Lucerne	
1.85	Oats	
1.8	Vetch	
1.6	Faba bean	
1.25	Field pea, lupin	
<1.0	Lentil, chickpea	Sensitive

Source: PIRSA.

Nut grass (Figure 13) is often a good indicator of increased salt levels.

Figure 13: Nut grass growing in saline soil.

1.8.4 Soil chloride levels

Ferguson *et al.* (2006) found, when conducting the GRDC SIP08 Subsoil Constraints Project (NSW DPI) that soil chloride levels >600 mg/kg reduced root growth in crops such as chickpea, lentil and linseed. Soil analysis should be conducted to identify levels of chloride and at what depth it changes.

Thresholds for chloride concentration in soil and yield reductions differ between crops (Table 11 and Table 12).

Table 11: Thresholds for chloride concentration in soil (mg/kg)

Crop	10% yield reduction	50% yield reduction
Bread wheat	700	1500
Durum wheat	600	1200
Barley	800	1500
Canola	1200	1800
Chickpea	600	1000

Source: Qld Natural Resources and Water Bulletin.

Table 12: 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	600–1200 mg Cl/kg	>1200 mg Cl/kg
<500 mg Na/kg	500–1000 mg Na/kg	>1000 mg Na/kg

Source: Qld Natural Resources and Water Bulletin.

Agronomic practices and crop choice

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 are low salinity constraints.

Low constraints of Na and Cl (<600 mg Cl/kg, <500 mg Na/kg in top 1 m soil depth):

- Cereal–legume rotations are possible.
- Canola can be grown.
- Cereal diseases must be managed.
- Opportunity cropping to utilise available soil water can be tried.

Medium constraints of Na and Cl (600–1200 mg Cl/kg, 500–1000 mg Na/kg in top 1 m soil depth): tolerant crops should be grown (wheat, barley, canola):

- Consider tolerant crop varieties.
- The more tolerant of the pulses (vetch, faba bean possibly lupin and field pea) will likely suffer yield penalties if grown.
- Match inputs to realistic yields.
- Cereal diseases must be managed.
- Avoid growing salt-susceptible pulses (lentil, chickpea) 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 (>1200 mg Cl/kg, >1000 mg Na/kg in top 1 m soil depth):

- Avoid growing crops or grow tolerant cereals.
- Match inputs to realistic yields.
- Consider alternative land use to cropping (e.g. saline-tolerant forages, pastures).

1.8.5 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.

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.

1.8.6 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 N fixation, so deficiency of these trace elements can lead to poor nodulation.

Refer to Table 13 for nutrient constraints based on soil pH.

Table 13: Soil classifications for pH (1:5 soil:water).

Increasing acidity							Increasing alkalinity
Acidic				Neutral	Alkaline		
3	4	5	6	7	8	9	10
Toxicity of : Aluminium (Al) Manganese (Mn) Iron (Fe)			**Ideal pH Range** for plant growth			Toxicity of : Sodium (Na) Boron (Bo) Bicarbonate (HCO3)	
Deficiency of: Magnesium (Mg) Calcium (Ca) Potassium (K) Phosphorus (P) Molybdenum (Mo)						Deficiency of: Fe Zinc (Zn) Mn Copper (Cu) P	

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.

Monocultures of cereals can lead to a build-up of cereal diseases. [20]

1.9 Herbicide residues in soil

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.

Weed burden in the new crop will depend on the seed set from last year and residual herbicide efficacy.

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. Leaves become off-colour, roots may be clubbed, and plants stop growing and eventually die. Refer to the labels for recommendations on plant-back periods for pulses following use of any herbicides. Be especially wary under conditions of limited rainfall since herbicide application.

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 (Figures 14, 15). In more severe cases, bare areas are left in the crop where this herbicide had been used, in some cases more than 5 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, particularly during the summer fallow or weed-control period (after November).

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

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.

Figure 14: Effect of Tordon® soil residues affecting faba bean. Note the stem distortion and severe leaf curl. (Source: Grain Legume Handbook (2008)).

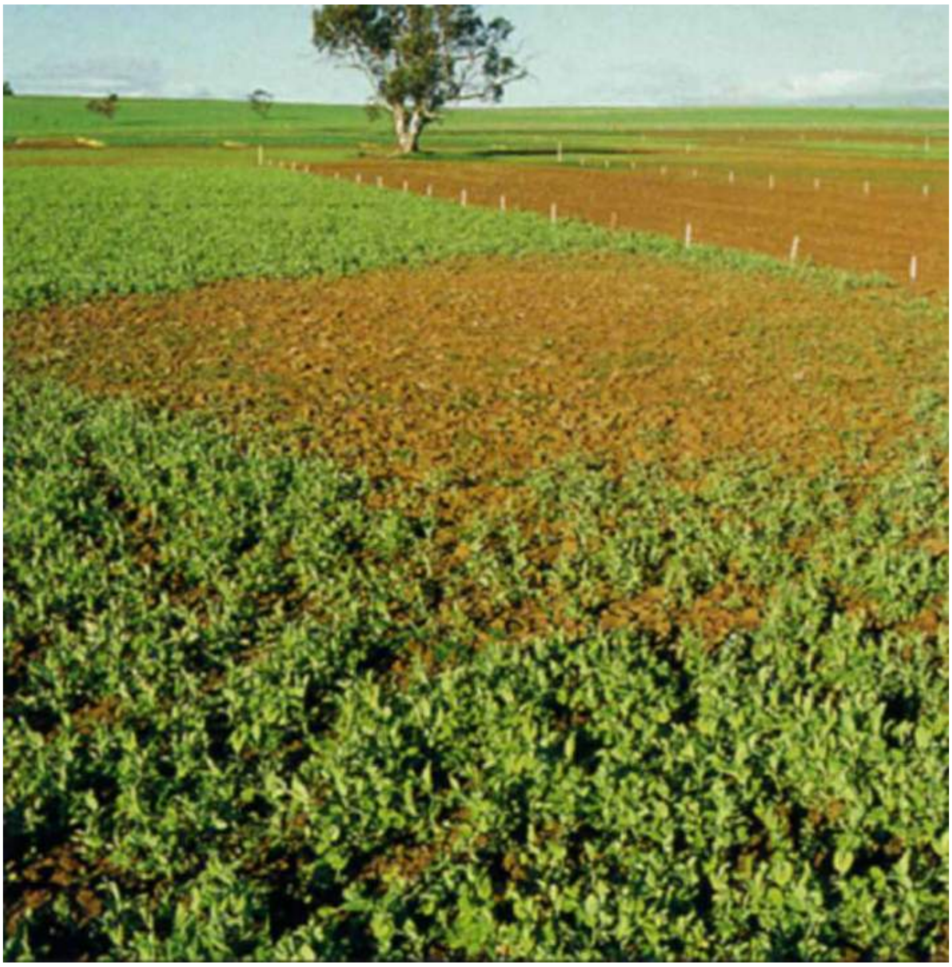


Figure 15: Tordon® spot-spray effect. Plants in the affected area are stunted. (Source: Grain Legume Handbook (2008).

1.9.1 Sulfonylurea residues, Group B

Sulfonylurea products include:

- metsulfuron (Ally®, Associate®, Lynx®)
- thifensulfuron plus metsulfuron (Harmony®M)
- sulfosulfuron (Monza®)
- chlorsulfuron (Glean®, Lusta®, Logran®)

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.

An application rate of 20 g Glean®/ha equates to an initial soil residue level of 10 parts per billion (ppb) in the top 10 cm soil.

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 (Figure 16).

Figure 16: Yellowing of new growth (left) and plant stunting (right). (Photo: A. Storrie)

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.

1.9.2 Imidazolinone (imi) residues, Group B

Imidazolinone products include:

- imazapic + imazapyr (Midas®, OnDuty®)
- imazamox + imazapyr (Intervix®)
- imazapic (Flame®)
- imazethapyr (Spinnaker®, various imazethapyrs)
- imazamox (Raptor®)

Figure 17: Spinnaker injury to the emerging new chickpea growth. (Photo: G. Cumming, Pulse Australia)

Imazethapyr (e.g. Spinnaker®) is registered for use in chickpeas in Victoria and South Australia, but can be damaging (Figure 17). 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 150-200mm 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.

Breakdown of imi products occurs by hydrolysis, and is favoured by warm, moist conditions in neutral to acid soils. Residues will tend to persist for longer under alkaline and/or dry conditions. Persistence of imi residues is greater for Intervix® and Midas® or OnDuty® than for Flame®.

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)

- lentil
- conventional canola
- safflower
- oats

Strategy

Avoid using imi products on very high pH soils (pH >8.5) 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.

1.9.3 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 (Figure 18). Products include: Gesaprim®, Nutrazine®, Farmozine®, various atrazines.

Atrazine significantly increases the frost sensitivity of the crop. Risk of damage increases where there are low levels of subsoil moisture (e.g. double-crop situations). Crops in this situation are largely surface-rooted and vulnerable to damage when there is herbicide recharge after each rainfall event.

Figure 18: 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)

All the evidence suggests that high rates of atrazine (>2.5 L atrazine/ha) should be avoided if considering double-cropping to chickpea after a summer forage or grain crop of sorghum or maize.

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 (e.g. when double-cropping chickpeas after cotton).

Highly susceptible indicator weeds

- mintweed (turnip, mustard, radish)
- brassicas
- black pigweed

Strategy

Avoid using heavier rates of atrazine (rate per ha depending on soil type) for summer grain or forage crops (e.g. sorghum, maize) if the intention is to follow with chickpeas in autumn–winter. Revise the strategy completely on highly alkaline black earths if high rates have been used in summer crops and dry conditions have been experienced.

1.9.4 Group I

Products include:

- 2,4-D products (amines, esters)
- dicamba (e.g. Cadence®)
- triclopyr (e.g. Garlon®)
- fluroxypyr (e.g. Starane®)

Residues of 2,4-D persist for a relatively short period, and they can be overlooked. Figure 19 depicts residual damage from 2,4-D. Table 14 shows the plant-back period 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 re-cropping interval was not the cause; rather, the damage was due to not having received the minimal rainfall requirement of 15 mm before this period commenced.

Figure 19: Residual 2,4-D damage, showing narrowing and thickening of leaflets on younger growth. (Photo: J. Flemming, NSW DPI)

Table 14: Chickpea plant-back intervals and conditions after spikes in Group I knockdown herbicides

Active Ingredient	Products	Rates (/ha)	Period	Comments
2,4-D	2,4-D amine (625 g/L)	Up to 0.56 L	7 days	At least 15 mm of rain must fall prior to commencement of the plant-back period
	2,4-D ester (800 g/L)	Up to 0.35 L		
	Baton® (800 g/kg) (amine)	Up to 0.4 kg		
	2,4-D amine (625 g/L)	0.56–1.1 L		
	2,4-D ester (800 g/L)	0.35–0.7 L	14 days	
	Surpass® (300 g/L) (amine)	1.1–2.3 L		
	Baton® (800 g/kg) (amine)	0.4–0.9 kg		
	2,4-D amine (625 g/L)	1.1–17 L		
	2,4-D ester (800 g/L)	0.7–1.1 L	21 days	
	Surpass® (300 g/L) (amine)	2.3–3.4 L		
	Baton® (800 g/kg) (amine)	0.9–1.3 kg		
	Cadence®	140 g		
Dicamba (700 g/kg)	Cadence®	200 g	Not determined	When applied to dry soil, at least 15 mm of rainfall is required prior to commencement of the plant-back period
	Dicamba 500	400 g	21 days	
		200 mL	28 days	
Dicamba (500 g/L)	Dicamba 500	280 mL	Not determined	
	Garlon® 600, Invader® 600	560 mL	21 days	
	Safari® 600	Up to 160 mL	28 days	
Triclopyr (600 g/L)	Starane® 200, Flagship®	Up to 375 mL	7 days	
Fluroxypyr (200 g/L)			7 days	

1.9.5 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 Tordon® products in sorghum crops if you are considering the option of double-cropping back to chickpeas.

Avoid using Lontrel® or Grazon® DS in the fallow period prior to chickpeas. [21]

More information

G Wockner, D Freebairn (2016), Commonly asked questions about soil water and soil management

D Freebairn (2016), Improving fallow efficiency

K Verburg, B Cocks, T Webster, J Whish (2016), Methods and tools to characterise soils for plant available water capacity (Coonabarabran)

D Freebairn (2016), SoilWaterApp – a new tool to measure and monitor soil water

1.10 Soil moisture

1.10.1 Dryland

Desi chickpea varieties should be grown only in areas where the rainfall is >350 mm. Sowing is best carried out from early May to early June, with early sowing recommended for the lower rainfall areas.

Kabuli chickpea varieties are later maturing and should be grown only in areas where rainfall is >450 mm. [22]

Mild winter conditions can promote rapid vegetative growth, resulting in valuable soil moisture being used to grow high-biomass crops. [23]

With the possible exception of parts of central-western NSW on lighter soils, soil moisture storage during fallows and subsequent extraction and use during a crop season are at least as important as in-season rainfall for achieving a profitable yield result in most of the northern grains region.

The 2013 winter was an extreme example, when many crops in Queensland and north-eastern and north-western NSW were successfully grown on little or no effective in-crop rainfall. Although not always to that extent, subsoils and the root activity in them are key to success in most northern cropping seasons.

The frequency of such seasons obviously varies (winter *v.* summer, and between regions), and a research team led by Mike Bell, Queensland Alliance for Agriculture and Food Innovation (QAAFI), has used the climatic record from representative sites across the northern grains region to estimate the frequency of occurrence.

The researchers have also used APSIM (Agricultural Production Systems Simulator) to generate yield distributions for these seasons at high (240 mm) and low (120 mm) soil plant-available water storage capacity (PAWC) and different starting profile moisture contents (full, two-thirds full or one-third full). These simulations were used to estimate average yields of the four key crops (wheat, chickpea, mungbean and sorghum) in each season type, with these averages used to estimate the potential yields, from which losses due to phosphorus (P) deficiency can be calculated. These data are shown for wheat and chickpea, sown on a two-thirds full moisture profile in soils with a PAWC of 120 or 240 mm (Table 15).

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

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

23 K Moore *et al.* (2014) Phytophthora tolerance in chickpea varieties. GRDC Update Papers 4 March 2014, http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Phytophthora-tolerance-in-chickpea-varieties

Table 15: Frequency of occurrence of different season types (%), and the simulated crop yields (kg/ha) for wheat and chickpeas under those seasonal conditions for different production centres and soil types, with plant-available water capacity of 120 or 240 mm.

Location		Dry starts				Wet start, dry finish				No serious water stress			
		120 mm		240 mm		120 mm		240 mm		120 mm		240 mm	
		%	Yield	%	Yield	%	Yield	%	Yield	%	Yield	%	Yield
Emerald	Wheat	40	850	40	2530	38	1470	12	2370	22%	2780	48	3280
	Chickpea	28	540	28	1730	48	930	7	1470	23%	2140	65	2540
Dalby	Wheat	12	980	12	2260	69	1610	43	2890	19%	3860	45	5150
	Chickpea	7	660	7	1580	49	890	13	1650	44%	2330	80	2770
Goondiwindi	Wheat	10	1070	10	2520	66	1700	27	3040	24%	4430	63	4970
	Chickpea	5	720	5	2060	48	1060	6	1840	47	2390	90	2800
Gunnedah	Wheat	6	880	6	2560	56	2080	30	3340	39	4390	65	5490
	Chickpea	5	990	5	2140	36	1130	6	2930	59	2460	90	2930
Walgett	Wheat	11	775	11	2040	75	1670	43	2840	14	3840	46	4790
	Chickpea	9	630	9	1750	59	1030	16	1650	32	2130	75	2570
Condobolin	Wheat	6	780	6	2040	75	1940	51	3210	19	4640	44	5680
	Chickpea	4	590	4	1550	56	1110	11	1660	40	2240	85	2604

The data are derived from profiles assumed two-thirds full at sowing, with similar data available for sorghum and mungbeans from spring and summer sowing windows. These yields would be achieved if there were no nutrient limitations.

More information

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Changing-nutrient-management-strategies-in-response-to-declining-background-fertility

Some important points about this analysis are:

- The frequency of Type 1 seasons (dry starts) is relatively low (5–10% of years), except in the Central Highlands of Queensland, where it ranges from 30 to 40% of years.
- There is a strong interaction between Type 2, late-stress seasons and soil type/PAWC, with the higher PAWC reducing the frequency of late-stress seasons by an average of 30% (wheat) to 40% (chickpeas). Average yields in those less frequent Type 2 seasons were still 800 (chickpeas) to 1200 (wheat) kg/ha higher in the high PAWC soils.
- Similar effects were evident in the summer crops (not shown), although the frequency of Type 1 seasons was much higher (average of 40% for sorghum and 21% for mungbean) than in winter crops. Higher PAWC reduced the average frequency of late-stress summer seasons by an average of 16% (sorghum) to 30% (mungbeans), with average yields in those late-stress seasons 1550 (sorghum) and 500 (mungbeans) kg/ha higher in the high PAWC soil. [24]

1.10.2 Irrigation

Soils must be well drained to reduce risk of waterlogging. Be aware of and monitor subsoil constraints that could limit yield potential.

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 is 1 tonne grain per megalitre (ML) water supply (per ha).

Pre-sowing irrigation may be optional, depending on stored soil moisture following summer rainfall. Timing of the first in-crop irrigation is critical and must be pre-flowering, when PAW reaches 30–40% depletion. Sufficient moisture must be supplied to cover the flowering period. However, waterlogging an already stressed crop at flowering can cause severe yield loss or actual plant death (Figure 20). It is imperative to get the water on and off the paddock as quickly as possible when irrigating chickpeas.

[24] M Bell *et al.* (2014) Changing nutrient management strategies in response to declining background fertility. GRDC Update Papers 4 March 2014, http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Changing-nutrient-management-strategies-in-response-to-declining-background-fertility

Figure 20: An aerial shot of crops west of Moree, NSW, in 2012 shows waterlogging that caused plant deaths.

More information

http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0004/176053/North-Irrig-surface-chickpeas-2012.pdf

Irrigation management tips:

- Filling the soil moisture profile at sowing time is important, and pre-irrigation has been a recommended practice. However, in recent dry seasons, growers have chosen to water-up, enabling them to incorporate pre-emergent herbicides. Ensure that seed placement allows at least 7 cm of soil above the seed if using Balance® or simazine and that the soil surface is left flat to prevent herbicide leaching into the plant furrow.
- Generally, in-crop irrigation should start early when there is a soil moisture deficit of 30–40 mm (or 60–70% of field capacity). Soil moisture deficit is more important in scheduling irrigations than plant growth stage.
- Irrigations should also commence prior to flowering to prevent impacts of moisture stress and high temperatures on grain size, quality and yield. This is particularly important with Kabuli types, where premiums are paid for larger seed sizes.
- The higher clay content or soil bulk density, the higher is the risk of waterlogging from slow water infiltration and subsequent slow draining. It is a greater risk to irrigate these soils once flowering and podding has commenced. If in doubt, do not water. [25]
- If the ground has cracked open irrigation should be avoided as this allows water to enter the root zone and cause waterlogging, leading to flower abortion and risking plant death.

More information

L Lake, V Sadras (2015), The critical period for yield determination in chickpea

1.11 Yield and targets

1.11.1 Seasonal outlook

The online tool CropMate was developed by NSW DPI and can be used in pre-season planning to analyse average temperature, rainfall and evaporation (Figure 21). It provides

25 NSW DPI (2012) Surface irrigated chickpeas. Farm Enterprise Budget Series. NSW Department of Primary Industries http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0004/176053/North-Irrig-surface-chickpeas-2012.pdf

seasonal forecasts and information about influences on climate, such as the impact of Southern Oscillation Index (SOI) on rainfall.

Download CropMate from the App Store on iTunes at: https://itunes.apple.com/au/app/cropmate-varietychooser/id476014848?mt=8

Figure 21: Screen shot of CropMate app. (Photo: NSW DPI)

Queensland Alliance for Agriculture & Food Innovation produces regular, seasonal outlooks for wheat producers in Queensland. These high-value reports are written in an easy-to-read style and are free. Download the 'Seasonal Crop Outlook—wheat, October 2015'.

For tips on understanding weather and climate drivers, including the SOI, visit the Climate Kelpie website. Case studies of 37 farmers across Australia recruited as 'Climate Champions' as part of the Managing Climate Variability R&D Program can also be accessed at the Climate Kelpie website.

Australian CliMate is a suite of climate analysis tools delivered on the web, and on iPhone, iPad and iPod Touch devices. CliMate allows you to interrogate climate records on 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 at: https://itunes.apple.com/au/app/australian-climate/id582572607?mt=8 or visit http://www.australianclimate.net.au

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 relative to previous seasons, based on heat sum?

More information

http://www.qaafi.uq.edu.au/seasonal-crop-outlook-wheat

http://www.climatekelpie.com.au/understand-climate/weather-and-climate-drivers/queensland#ElNino

http://www.climatekelpie.com.au/ask-a-farmer/climate-champion-program

- Is there any reason why my crop is not doing as well as usual because of below-average rainfall or radiation?
- Based on 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. [26]

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. [27]

More information

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/07/Seasonal-climate-outlook-improvements-changes-from-historical-to-real-time-data

www.australianclimate.net.au

1.11.2 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. [28]

More information

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/07/Impact-of-stored-water-on-risk-and-sowing-decisions-in-western-NSW

K Verburg, J Which (2016), Drivers of fallow efficiency: Effect of soil properties and rainfall patterns on evaporation and the effectiveness of stubble cover

Cover crops

During the 14-month-long fallow that arises when moving from summer to winter crops, stubble breakdown can denude the soil surface and leave it vulnerable to erosion. Cover crops of millet have been proposed as a solution, but this raises the question, how often is there sufficient water in the system to grow a cover crop without reducing the soil water reserves to the detriment of the following wheat crop?

An on-farm research approach was used to compare the traditional long fallow with a millet fallow in 31 commercial paddocks over 3 years. Each treatment was simulated using the simulation-modelling framework APSIM to investigate the outcomes over a longer timeframe and to determine how often a millet fallow could be successfully included within the farming system.

The trials showed that early-sown millet cover crops removed before December had no effect on wheat yield, but this was not true of millet cover crops allowed to grow through to maturity. Long-term simulations estimated that a spring cover-crop of millet would adversely affect wheat yields in only 2% of years if planted early and removed after 50% cover had been achieved. [29]

There are few options for the control of problem grass weeds such as ABYG and FTR while the millet crop is growing.

It has also been observed that in wet summers, *Fusarium* spp. fungus, which causes crown rot, can colonise and survive on some species of millet.

[26] Australian CliMate—Climate tools for decision makers, www.australianclimate.net.au

[27] 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

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

[29] JPM Whish, L Price, PA Castor (2009) Do spring cover crops rob water and so reduce wheat yields in the northern grain zone of eastern Australia? *Crop & Pasture Science* 60, 517–525.

HowWet?

HowWet? is a program that uses records from a nearby weather station to estimate how much PAW has accumulated in the soil and the amount of organic N that has been converted to an available nitrate during a fallow. HowWet? tracks soil moisture, evaporation, runoff and drainage on a daily time-step. Accumulation of available N in the soil is calculated based on surface soil moisture, temperature and soil organic carbon.

HowWet?

1. Estimates how much rain has been stored as plant-available soil water during the most recent fallow period;
2. Estimates the N mineralised as nitrate-N in soil; and
3. Provides a comparison with previous seasons.

This information aids the decision about what crop to plant and how much N fertiliser to apply.

Many grain growers are in regions where stored soil water and nitrate at planting are important in crop management decisions. This is particularly important to northern Australian grain growers with clay soils where stored soil water at planting can constitute a large part of a crop's water supply.

Questions this tool answers:

- How much longer should I fallow? If the soil is near full, perhaps the fallow can be shortened.
- Given my soil type and local rainfall to date, what is the relative soil moisture and nitrate-N accumulation over the fallow period compared with most years? Relative changes are more reliable than absolute values.
- Based on estimates of soil water and nitrate-N accumulation over the fallow, what adjustments are needed to the N supply? [30]

Inputs

- A selected soil type and weather station
- An estimate of soil cover and starting soil moisture
- Rainfall data input by the user for the stand-alone version of HowOften?

Outputs

- A graph showing plant-available soil water for the current year and all other years and a table summarising the recent fallow water balance
- A graph showing nitrate accumulation for the current year and all other years

Reliability

HowWet? uses standard water-balance algorithms from HowLeaky? and a simplified nitrate mineralisation based on the original version of HowWet? Further calibration is needed before accepting with confidence absolute value estimates.

Soil descriptions are based on generic soil types with standard organic carbon (C) and C/N ratios, and as such should be regarded as indicative only and best used as a measure of relative water accumulation and nitrate mineralisation. [31]

More information

http://www.australianclimate.net.au/About/HowWetN

1.11.3 Water-use efficiency

Water-use efficiency is the measure of a cropping system's capacity to convert water into plant biomass or grain. It includes the use of water stored in the soil and rainfall during the growing season.

30 Australian CliMate—How Wet/N, http://www.australianclimate.net.au/About/HowWetN

31 Australian CliMate—How Wet/N, http://www.australianclimate.net.au/About/HowWetN

Water-use efficiency 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).

Water is the principal limiting factor in rain-fed cropping systems in northern Australia. The objective of rain-fed cropping systems is to maximise the proportion of rainfall that crops use, and minimise water lost through runoff, drainage and evaporation from the soil surface and to weeds.

Rainfall is more summer-dominant in the northern region, and both summer and winter crops are grown. However, rainfall is highly variable and can range, during each cropping season, from little or no rain to major events that result in waterlogging or flooding.

Storing water in fallows between crops is the grower's most effective tool to manage the risk of rainfall variability, as in-season rainfall alone, in either summer or winter, is rarely enough to produce a profitable crop, especially with high levels of plant transpiration and evaporation.

Fortunately, many cropping soils in the northern region have the capacity to store large amounts of water during the fallow. [32]

The French–Schultz approach

In southern Australia, the French–Schultz model is widely used to provide growers with a benchmark of potential crop yield based on available soil moisture and likely in-crop rainfall.

In this model, potential crop yield is estimated as:

Potential yield (kg/ha) = WUE (kg/ha.mm) x [crop water supply (mm) – estimate of soil evaporation (mm)]

where crop water supply is an estimate of water available to the crop, i.e. soil water at planting plus in-crop rainfall minus soil water remaining at harvest.

In the highly variable rainfall environment in the northern region, it is difficult to estimate in-crop rainfall, soil evaporation and soil water remaining at harvest. However, this model may still provide a guide to crop yield potential (Table 16 and Table 17).

The French–Schultz model has been useful in giving growers performance benchmarks; where yields fall well below these benchmarks it may indicate something wrong with the crop's agronomy or a major limitation in the environment. There could be hidden problems in the soil such as root diseases, or soil constraints affecting yields. Alternatively, apparent underperformance could be simply due to seasonal rainfall distribution patterns, which are beyond the grower's control. [33]

Table 16: Typical parameters that could be used in the French–Schultz equation

Crop	WUE (kg/ha.mm)	Soil evaporation (mm)
Wheat	18	100
Chickpea	12	100
Sorghum	25	150

32 GRDC (2009) Water use efficiency—converting rainfall to grain. GRDC Fact Sheet Northern Region, http://www.grdc.com.au/~/media/607AD22DC6934BE79DEAA05DFBE00999.pdf

33 GRDC (2009) Water use efficiency—converting rainfall to grain. GRDC Fact Sheet Northern Region, http://www.grdc.com.au/~/media/607AD22DC6934BE79DEAA05DFBE00999.pdf

More information

https://dl.sciencesocieties.org/publications/meetings/download/pdf/2013am/78228

Table 17: Effect of soil water threshold for planting on system water-use efficiency (SWUE) and other system performance parameters.

System:		Conservative	Moderate	Aggressive
Planting threshold	mm	150	100	50
Number of crops		35	45	72
Crops/year		0.69	0.88	1.41
Total grain produced	t/ha	141	172	197
Average yield	t/ha	4.04	3.82	2.73
Average cover	%	40%	49%	55%
SWUE	kg/ha.mm	4.55	5.53	6.32
% rainfall ending up as:				
Transpiration		21%	26%	32%
Evaporation		56%	55%	55%
Run-off		18%	16%	11%
Drainage		5%	3%	2%

This table presents the results of a simulation modelling analysis for a cropping system at Emerald from 1955 to 2006

Challenging the French–Schultz model

Application of the French–Schultz model for the northern region has been challenged in recent times.

In the grain-belt of eastern Australia, rainfall shifts from winter-dominated in the south (South Australia, Victoria) to summer-dominated in the north (northern NSW and Queensland). The seasonality of rainfall, together with frost risk, drives the choice of cultivar and sowing date, resulting in a flowering time between October in the south and August in the north.

In eastern Australia, crops are therefore exposed to contrasting climatic conditions during the critical period for grain formation (i.e. a window of about 20 days before and 10 days after flowering, which affects yield potential and WUE).

Understanding how those climatic conditions affect crop processes and how they vary from north to south and from season to season can help growers and consultants to set more realistic target yields across sites, locations and seasons (Figure 22).

Researchers have analysed some of the consequences of the shift from winter to summer rainfall between southern and northern regions in terms of implications for management and breeding. They advise caution on the use of simple rules of thumb (French–Schultz) for benchmarking WUE, and discuss the importance of more integrative and dynamic modelling approaches to explore alternatives to increase WUE at the single-crop and whole farming systems level (i.e. $/ha.mm).

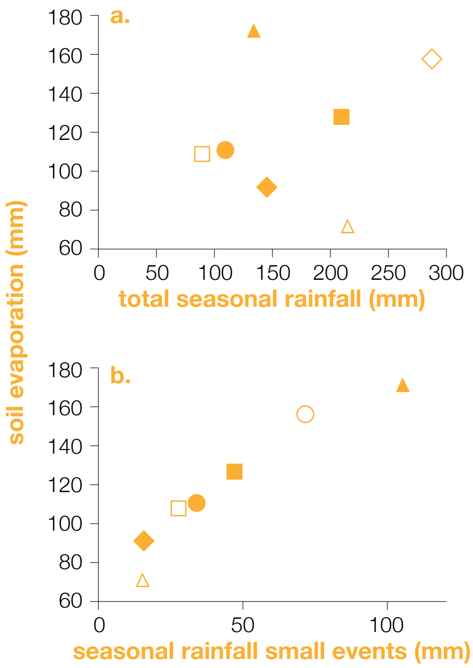


Figure 22: Simulated soil evaporation is (a) unrelated to seasonal rainfall and (b) closely related to rainfall in small events (i.e. equal to or below 5 mm).

More information

https://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2008/06/Farming-systems-design-and-water-use-efficiency-WUE-Challenging-the-French-Schultz-Wue-model

1.11.4 Double crop options

Chickpeas are a very good crop to double crop out of an early sorghum crop straight back into chickpea, avoiding the need for long fallow in wet summers. This is possible because chickpea needs less water than wheat.

1.12 Disease status of the 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 of the northern region cropping belt.

More information

http://www.pulseaus.com.au

http://www.grdc.com.au/Research-and-Development/Major-Initiatives/PBA/PBA-Varieties-and-Brochures

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.

Chickpea varieties in the northern region are susceptible to both species of *Sclerotinia*, and all varieties are susceptible to Botrytis grey mould (Table 18).

However, there are varying levels of resistances to Ascochyta blight and *Phytophthora* so growers should consider planting a variety with the highest levels of resistance to either or both. [34]

[34] GRDC (2011) What to consider before planting chickpeas. GRDC Media Centre 6 June 2011

Table 18: Resistance ratings of some northern region varieties to Phoma rabiei (Ascochyta rabiei), Phytophthora medicaginis and Botrytis cinerea

Variety	*Phoma rabiei*	*Phytophthora medicaginis*	*Botrytis cinerea*
PBA HatTrick⊕	MR/R	MR	S
Flipper⊕	MR	MS	S
PBA Boundary⊕	R/MR	MS	S
PBA Monarch⊕	MS	S	S
Yorker⊕	MS/MR	MR	S
Jimbour⊕	S	MS/MR	S
Kyabra⊕	S	MS	S
Genesis090	R	VS	VS
Genesis425	R	MS	S
Almaz⊕	MS/MR	VS	S

Resistance ratings are for situations of low–moderate disease pressure. In a season such as 2010 when repeated cycles of infection occur, even MR varieties can have yield-reducing levels of disease. M, moderately; V, very; R, Resistant; S, susceptible

1.12.1 Soil testing for disease

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 southern Australia and includes tests for:

- cereal cyst nematode
- take-all (*Gaeumannomyces graminis var. tritici* (Ggt) and *G. graminis var. avenae* (Gga))
- Rhizoctonia barepatch (*Rhizoctonia solani* AG8)
- crown rot (*Fusarium pseudograminearum*)
- root-lesion nematode (RLN) (*Pratylenchus neglectus* and *P. thornei*)
- stem nematode (*Ditylenchus dipsaci*)

More information

http://www.sardi.sa.gov.au/products__and__services/entomology/diagnostic_services/predicta_b

Crown Analytical Services

Northern region grain producers can access PreDicta B via Crown Analytical Services or agronomists accredited by the South Australian Research and Development Institute 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.

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.

1.13 Nematode status of the paddock

The RLN *Pratylenchus thornei* (Pt) is widespread in cropping soils through central and northern NSW. Although mainly considered an issue in wheat crops, Pt also infects chickpeas, with yield losses of 20–30% previously recorded in intolerant varieties. Chickpeas are also susceptible to Pt, which means that this nematode colonises the root systems and builds up numbers in the soil. This is especially an issue in the northern region where chickpeas remain the main winter break crop grown in rotation with winter cereals. However, chickpea varieties can vary in their levels of resistance to Pt; this is related to the extent to which they build up Pt 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 Pt population left in the soil following chickpeas, the greater is the potential for a negative impact on the yield of subsequent crops.

More information

http://www.soilquality.org.au/factsheets/root-lesion-nematode-in-queensland

Pratylenchus thornei costs the wheat industry AU$38 million annually. [35] Including the secondary species, *P. neglectus*, RLN is found in three-quarters of paddocks tested. [36]

1.13.1 Nematode testing of soil

It is important to have paddocks diagnosed for plant parasitic nematodes so that optimal management strategies can be implemented. Testing your farm will tell you:

- if nematodes are present in your paddocks and at what density
- 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. [37]

More information

http://www.sardi.sa.gov.au/products__and__services/entomology/diagnostic_services/predicta_b

http://www.daf.qld.gov.au/__data/assets/pdf_file/0010/58870/Root-Lesion-Nematode-Brochure.pdf

Crown Analytical Services

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.

A chickpea variety trial was conducted at Come-by-Chance, north-western NSW, in 2010 under the National Variety Trial (NVT) network funded by GRDC. The harvested plots were left intact and soil cores were taken in March 2011 to assess the effect of chickpea variety choice on the build-up of Pt in the soil under the 2010 crop. This type of testing determines the resistance of chickpea varieties to Pt.

Desi chickpea entries varied significantly in their effect on the build-up of Pt populations in the soil over the 2010 season (i.e. resistance level). Pt populations multiplied 1.8 times under the most resistant entry, CICA1009, and up to 8.4 times under the most susceptible entry, CICA0907. Variety choice can also have a significant impact on the build-up of Pt populations within the soil, with numbers about 2.3 times higher after the very susceptible variety Kyabra⚲ than after moderately susceptible varieties such as PBA Boundary⚲, Jimbour⚲ or PBA HatTrick⚲.

All current Desi chickpea varieties and advanced lines are susceptible to Pt, and they will build up soil populations within the rotation. However, variety choice can still influence the extent of build-up of Pt, because significant differences exist in the resistance of chickpea varieties to Pt.

More information

http://www.dpi.nsw.gov.au/agriculture/broadacre/guides/ngrt-results

As highlighted in this study, the NVT network is a valuable potential source of reliable field assessments of nematode resistance levels in varieties and near-release lines across a range of crop types.

Breeding programs need to focus on developing and releasing chickpea varieties with good levels of tolerance to Pt to limit yield impact on chickpea crops. However, released varieties also need to have improved levels of resistance to Pt to limit the build-up of this widespread pest within cropping systems in the northern region. [38]

[35] GM Murray, JP Brennan (2009) The current and potential costs from diseases of wheat in Australia. GRDC Report, https://www.grdc.com.au/~/media/B4063ED6F63C4A968B3D7601E9E3FA38.pdf

[36] K Owen, J Sheedy, N Seymour (2013) Root lesion nematode in Queensland. Soil Quality Pty Ltd Fact Sheet, http://www.soilquality.org.au/factsheets/root-lesion-nematode-in-queensland

[37] Queensland Primary Industries and Fisheries (2009) Root lesion nematodes—management of root-lesion nematodes in the northern grain region. Queensland Government, http://www.daf.qld.gov.au/__data/assets/pdf_file/0010/58870/Root-Lesion-Nematode-Brochure.pdf

[38] 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

1.13.2 Effects of cropping history on nematode status

Root-lesion nematode numbers build up steadily under susceptible crops and cause decreasing yields over several years. Yield losses >50% can occur in some wheat varieties, 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 2000 RLN/kg soil anywhere in the soil profile has the potential to reduce the grain yield of intolerant wheat varieties.

A tolerant crop yields well when high populations of RLN are present (the opposite is intolerance). A resistant crop does not allow RLN 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 wheat or 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 RLN is regularly updated in grower and DAFF planting guides. Note that crops and varieties have different levels of tolerance and resistance to Pt and *P. neglectus*.

Summer crops have an important role in management of RLN. Research shows when Pt 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. [39]

For more information on nematode management, see Section 8, Nematodes

1.14 Feral pests

Significant losses of plant populations have been caused by feral pigs seeking out the germinating seed from in the soil. In areas adjoining pig-infested scrub, damage can be significant enough to justify re-planting areas.

More information

'Management of root-lesion nematodes in the northern grain region': http://www.daf.qld.gov.au/__data/assets/pdf_file/0010/58870/Root-Lesion-Nematode-Brochure.pdf

More information

https://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/07/Summer-crop-decisions-and-root-lesion-nematodes

More information

http://www.daf.qld.gov.au/plants/field-crops-and-pastures/broadacre-field-crops/integrated-pest-management/help-pages/recognising-and-monitoring-soil-insects

http://www.pestgenie.com.au/

http://www.apvma.gov.au/

http://www.feral.org.au/

http://www.daf.qld.gov.au/plants/field-crops-and-pastures/broadacre-field-crops/vertebrate-pests

[39] 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

SECTION 2

Pre-planting

More information

http://www.grdc.com.au/Research-and-Development/National-Variety-Trials/Crop-Variety-Guides

http://www.daf.qld.gov.au/plants/field-crops-and-pastures/broadacre-field-crops/chickpeas

http://www.dpi.nsw.gov.au/agriculture/broadacre/guides/winter-crop-variety-sowing-guide

Pulse Australia (2016), Chickpea production: northern region

QDAF (2015), Varieties of chickpeas

Pulse Australia (2015), Chickpea variety choices for 2015

K Hobson, A Verrell, A George, M Nowland, J Duncan (2014), PBA Chickpea program – Evaluation in 2013 p 36

2.1 Choosing a variety

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

The availability of varieties resistant to Ascochyta blight now provides growers with low disease-risk options for growing chickpea in northern 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.

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 (e.g. PBA Boundary⦶, which is susceptible to Phytophthora root rot).

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.

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. [1]

2.2 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):

- Region 1, low rainfall tropical
- Region 2, medium rainfall, subtropical
- Region 3, low rainfall, subtropical
- Region 4, medium–high rainfall Mediterranean–temperate
- Region 5, low–medium rainfall Mediterranean–temperate

1 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

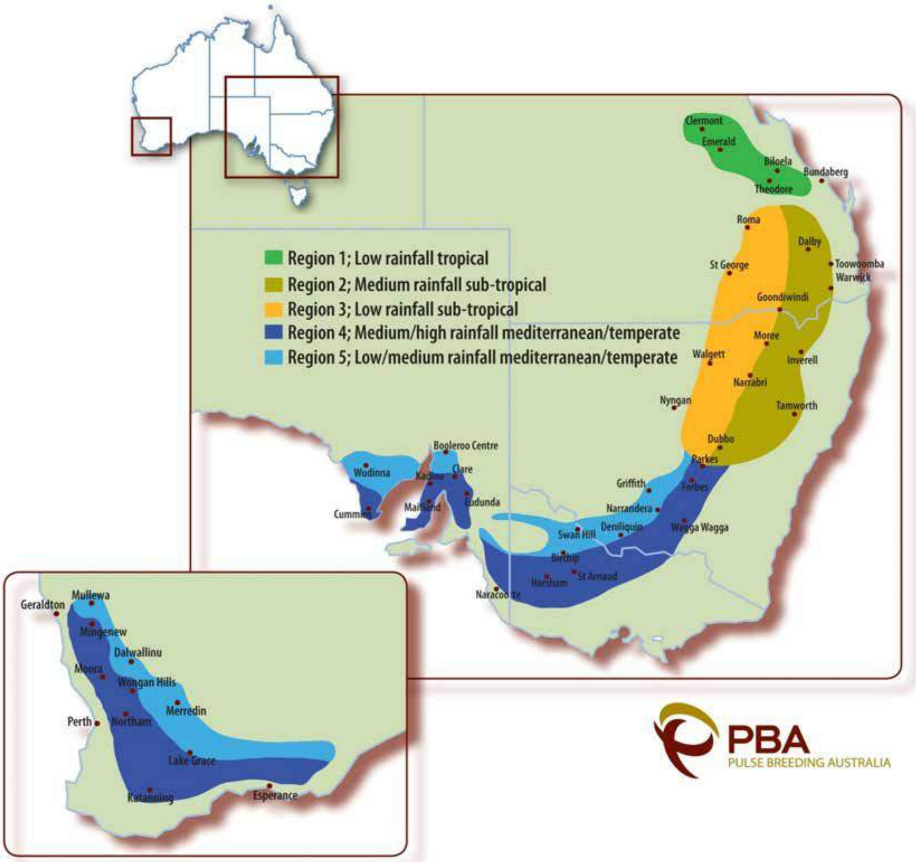


Figure 1: Five Australian chickpea-growing areas based on rainfall and geographic location.

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.

There have been variety releases specific for central Queensland (PBA SeamerⒹ, PBA PistolⒹ and MotiⒹ), southern Queensland and northern New South Wales (PBA HatTrickⒹ and PBA BoundaryⒹ).

The area of adaptation is specified for each variety so that potential users are aware of their best fit. [2]

More information

http://www.grdc.com.au/uploads/documents/PBA%20HatTrick%20-%20chickpea.pdf

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Chickpea-varieties-selecting-horses-for-courses

[2] 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

More information

http://www.nvtonline.com.au/variety-brochures/

http://www.grdc.com.au/Research-and-Development/Major-Initiatives/PBA/PBA-Varieties-and-Brochures

2.3 Evaluation of yield potential

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, www.nvtonline.com.au, as well as from the specific Pulse Variety Management Package (VMP) brochure at http://www.grdc.com.au/Research-and-Development/Major-Initiatives/PBA/PBA-Varieties-and-Brochures

Long-term yields can be represented in several different ways but are typically displayed either as site-specific, averaged over multiple years (Figure 2), or for each year averaged over multiple sites for a region (Table 1). All trial sites are disease-free. [3]

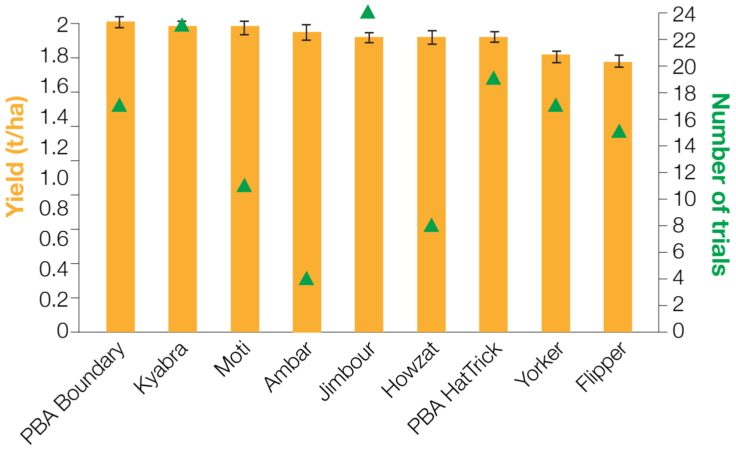


Figure 2: National Variety Trials long-term yield report: chickpea—Desi—SWQ—Billa Billa (2005–12). (Source: NVT)

Table 1: Long-term Desi chickpea yields in north-eastern Australia as a percentage of PBA HatTrick, 2006–10

Region 2, R2, Central/North-Western Slopes (NSW) and Darling Downs (Qld); Region 3, R3, Central/North-Western Plains (NSW) and Western Downs/Maranoa (Qld)

Variety	2010		2009		2008		2007		2006	
	R2	R3	R2	R3	R2	R3	R2	R3	R2	R3
Flipper	95	86	97	92	96	90	97	86	96	89
Jimbour	72	78	104	103	99	96	103	102	102	100
Kyabra	78	72	106	105	99	96	105	104	106	–
PBA Boundary	109	104	104	102	109	105	106	106	106	109
PBA HatTrick	100	100	100	100	100	100	100	100	100	100
Yorker	88	98	100	96	98	89	92	88	94	91
PBA HatTrick (t/ha)	2.12	2.28	1.78	1.56	2.14	1.96	1.27	1.10	1.86	1.68

Source: Pulse Breeding Australia and National Variety Trials.

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

More information

http://www.dpi.nsw.gov.au/agriculture/broadacre/guides/winter-crop-variety-sowing-guide

http://www.grdc.com.au/Research-and-Development/Major-Initiatives/PBA/PBA-Varieties-and-Brochures

https://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2010/05/Chickpeas-In-2010-PBA-Hattrick-Performance-And-Management

L Lake, V Sadras (2016). The critical period for yield determination in chickpea

L Jenkins, A Verrell (2015). Chickpea and mungbean research to maximise yield in northern NSW

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. [4]

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. [5]

2.3.1 Yielding ability

Desi types

Ambar⊕. Resistant (R) to Ascochyta, similar to Genesis™ 509 and Genesis™ 090, superior to PBA HatTrick and PBA Boundary; susceptible (S) to Phytophthora root rot, so not recommended for northern NSW. Limited evaluation in southern NSW. Developed by DAFWA and UWA from germplasm bred by NSW DPI. Marketed by Heritage Seeds. An EPR of $4.40/tonne applies.

Flipper⊕. Moderately resistant–moderately susceptible (MR–MS) to Ascochyta, less resistant than PBA HatTrick and PBA Boundary; MS to Phytophthora, less resistant than PBA HatTrick; S to viruses. Tall, erect variety with very good lodging resistance and medium sized seed. Bred by NSW DPI; commercial partner is Seednet with seed available through Seednet agents. An EPR of $3.30/tonne applies.

Jimbour. Susceptible to Ascochyta. Suited to areas where Ascochyta is not considered a major threat and experience shows that the disease can be managed in susceptible varieties; MS–MR to Phytophthora. Bred by DAF Qld, commercialised by Mt Tyson seeds. No EPR applies.

Kyabra⊕. Susceptible to Ascochyta – suited to areas where Ascochyta is not considered a major threat and experience shows that the disease can be managed in susceptible varieties; MS to Phytophthora; S to Botrytis grey mould. Larger seed size and superior grain quality for the whole seed market compared with other current varieties. Bred by DAF Qld and NSW DPI; commercial partner is Heritage Seeds. A seed royalty applies to all seed sales of Kyabra; no EPR applies.

Neelam⊕. Resistant to Ascochyta, similar to Genesis™ 509 and Genesis™ 090, superior to PBA HatTrick and PBA Boundary; S to Phytophthora root rot, so not recommended for northern NSW. Limited evaluation in southern NSW. Developed by DAFWA and UWA from germplasm bred by DEDJTR Victoria. Marketed by Heritage Seeds. An EPR of $4.40/tonne applies.

PBA Boundary⊕. Moderately resistant to Ascochyta, superior to PBA HatTrick; S to Phytophthora, less resistant than PBA HatTrick and only suitable for paddocks with a low Phytophthora risk. Highest yielding variety across chickpea growing regions of northern NSW and southern QLD. Lower yielding than PBA Slasher in southern NSW, but a suitable option if a tall, erect plant type is required. Mid-season maturity, equivalent to PBA HatTrick. Medium sized desi seed suited to the human consumption market. Developed by Pulse Breeding Australia (PBA). Marketed by Seednet with seed available through Seednet agents. An EPR of $4.40/tonne applies.

4 P Matthews, Don McCaffery, L Jenkins (2014) Winter crop variety sowing guide 2014. NSW DPI, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0011/272945/Winter-crop-variety-sowing-guide-2014.pdf

5 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

Figure 3: PBA Boundary⦶ has better Ascochyta blight resistance than PBA HatTrick⦶ but is more susceptible to Phytophthora root rot. (Photo: Gordon Cumming, Pulse Breeding Australia)

PBA HatTrick⦶. Moderately resistant to Ascochyta, superior to Flipper; MR to Phytophthora, more resistant than Jimbour, but less than Yorker. High-yielding variety across chickpea growing regions of northern NSW and southern Qld, recommended and suited to areas north of Parkes. Tall, erect plant type with mid-season maturity, equivalent to Jimbour. Medium sized desi seed suited to the human consumption market. Developed by Pulse Breeding Australia (PBA). Marketed by Seednet with seed available through Seednet agents. An EPR of $4.40/tonne applies.

Figure 4: PBA HatTrick⦶ is moderately resistant–resistant to Ascochyta blight. (Photo: Gordon Cumming, Pulse Breeding Australia)

PBA Seamer⦶ is an improved desi chickpea for the northern region with the highest available Ascochyta blight resistance rating (rated R). It is broadly adapted from central NSW to central Queensland, with significantly higher grain yield than all current varieties in high disease years. PBA Seamer⦶ has a semi-erect plant type with superior lodging resistance to PBA HatTrickA and PBA Boundary⦶. PBA Seamer⦶ has improved seed quality with larger seed size than PBA HatTrick⦶ and higher dhal milling yield than all current varieties in southern QLD and northern NSW.

PBA Maiden⦶. Moderately resistant to Ascochyta, less than PBA Slasher; S to Phytophthora root rot, so not recommended for northern NSW. Semi-spreading plant type with mid-season maturity, similar to PBA Slasher. Large sized desi for southern environments with a yellow-tan seed coat suited to whole seed markets. Developed by Pulse Breeding Australia (PBA). Marketed by Seednet with seed available through Seednet agents. An EPR of $4.40/tonne applies.

PBA Slasher⦶. Resistant to Ascochyta, similar to Genesis™ 509 and Genesis™ 090, superior to PBA HatTrick and PBA Boundary; S to Phytophthora root rot, so not recommended for northern NSW. High-yielding variety across all southern and western Australian chickpea growing regions, recommended and suited to areas south of Parkes. Semi-spreading plant type with mid-season maturity, similar to Howzat. Medium sized desi with tan–brown seed coat suitable for the whole and split seed markets. Developed by Pulse Breeding Australia (PBA). Marketed by Seednet with seed available through Seednet agents. An EPR of $4.40/tonne applies.

PBA Striker⦶. Moderately resistant to Ascochyta, less than PBA Slasher; S to Phytophthora root rot, so not recommended for northern NSW. High-yielding variety in short season environments in southern and western Australian chickpea growing regions. Semi-spreading plant type with earlier flowering and maturity than PBA Slasher. Medium sized desi with tan–brown seed coat suitable for the whole and split seed markets. Developed by Pulse Breeding Australia (PBA). Marketed by Seednet with seed available through Seednet agents. An EPR of $4.40/tonne applies.

Yorker⦶. Moderately susceptible to Ascochyta, inferior to PBA HatTrick and PBA Boundary; MR to Phytophthora, better than PBA HatTrick. Suited to drier areas where Phytophthora rather than Ascochyta is the greater risk. Yorker is sensitive to Balance® herbicide (see Weed control in the next column). Bred by NSW DPI; commercial partner is Seednet with seed available through Seednet agents. An EPR of $3.30/tonne applies.

Kabuli types

Almaz⦶. Moderately susceptible to Ascochyta, inferior to Genesis™ 090 and Genesis™ 425; S to Phytophthora. Medium seed size, 8–9 mm. Introduced from ICARDA, Syria and selected by DAFWA. Commercial partner is COGGO Group. Contact Seednet in eastern Australia for seed orders. An EPR of $7.15/tonne applies.

Genesis™ 090. Resistant to Ascochyta, equal to Genesis™ 509; broadly adapted; VS to Phytophthora, suited only to areas with a low Phytophthora risk. Seed size is smaller than Almaz, predominantly 7–8 mm. Introduced from ICARDA, Syria and selected by DEDJTR Victoria. Marketed by Australian Agricultural CroP.Technologies. An EPR of $5.00/tonne applies.

Genesis™ 114. Moderately susceptible to Ascochyta, inferior to Genesis™ 090 and Genesis™ 425; S to Phytophthora. Medium seed size similar to Almaz, predominantly 8–9 mm. Introduced from ICARDA, Syria and selected by DEDJTR Victoria. Excellent harvestability with an erect plant habit and good lodging resistance. Marketed by Australian Agricultural CroP.Technologies. An EPR of $5.50/tonne applies.

Genesis™ 425. Resistant o Ascochyta, superior to Almaz, and equal to Genesis™ 090. The least susceptible kabuli variety to Phytophthora but a susceptible rating means it will sustain economic yield loss in high risk Phytophthora situations. Higher yielding than Almaz, but lower yielding than Genesis™ 090. Seed size is smaller than Almaz, but slightly larger than Genesis™ 090 (predominantly 8 mm). Genesis™ 425 has shown some sensitivity to Balance® in northern NSW trials and herbicide screening trials in South Australia. Introduced from ICARDA, Syria and selected by DEDJTR Victoria and NSW DPI. Marketed by Australian Agricultural CroP.Technologies. An EPR of $5.50/tonne applies.

Genesis™ Kalkee. Moderately susceptible to Ascochyta, inferior to Genesis™ 090 and Genesis™ 425; S to Phytophthora. Larger seed size than Almaz and Genesis™ 114, predominantly 9 mm. Introduced from ICARDA, Syria and selected by DEDJTR Victoria and NSW DPI. Yield is similar to Genesis™ 114 and Almaz in northern and southern NSW. Excellent harvestability with an erect plant habit and good lodging resistance. Marketed by Australian Agricultural CroP.Technologies. An EPR of $5.50/tonne applies.

PBA Monarch⦶. Moderately susceptible to Ascochyta, inferior to Genesis™ 090 and Genesis™ 425; S to Phytophthora. Early flowering and early maturing. Medium seed size, 8–9 mm, similar to Almaz. Highest yielding medium sized kabuli chickpea. Semispreading plant type, which can be prone to lodging. Developed by Pulse Breeding Australia (PBA). Marketed by Seednet with seed available through Seednet agents. An EPR of $7.15/tonne applies. [6]

Characteristics and disease ratings for these varieties are summarised in Tables 2 and 3.

Table 2: Chickpea variety characteristics

Variety	Plant height	Lodging resistance	100 seed weight (g)	Maturity	North: Yield as a % of PBA HatTrick 2011–2015 East 1.94 t/ha	North: West 1.55 t/ha	South: Yield as a % of PBA Slasher 2011–2015 East 1.56 t/ha	South: West 1.36 t/ha
Desi Types								
Ambar	MS	VG	16	E	n.d.	n.d.	93 (3)	95 (3)
Flipper	T	VG	18	M–L	n.d.	94 (5)	n.d	n.d
Howzat	M	M	21	M	n.d.	n.d.	95 (5)	95 (5)
Jimbour	T	VG	20	M	101 (13)	101 (35)	n.d.	n.d
Kyabra	T	VG	26	M	106 (10)	109 (28)	n.d.	n.d.
Neelam	MT	VG	17	M	n.d.	n.d.	99 (3)	99 (3)
PBA Boundary	T	G	19	M	102 (13)	103 (35)	94 (5)	95 (5)
PBA HatTrick	T	G	20	M	100 (13)	100 (35)	91 (5)	92 (5)
PBA Maiden	MS	M	24	M	n.d.	n.d.	97 (5)	99 (5)
PBA Slasher	MS	M	18	M	n.d.	n.d.	100 (5)	100 (5)
PBA Striker	MS	M	21	E	n.d	n.d	100 (5)	102 (5)
Yorker	M	G	21	M–L	n.d	95 (5)	n.d.	n.d
Variety	**Plant height**	**Lodging resistance**	**100 seed weight (g)**	**Maturity**	**Yield as a % of Almaz 2011–2015 East 2.03 t/ha**	**West 1.52 t/ha**	**Yield as a % of Genesis™ 090 2011–2015 East 1.35 t/ha**	**West n.d.**
Kabuli types								
Almaz	MT	G	41	L	100 (5)	100 (17)	93 (5)	n.d.
Genesis™ 090	M	G	30	M–L	104 (5)	109 (17)	100 (5)	n.d.
Genesis™ 114	T	VG	39	M–L	102 (5)	103 (11)	91 (5)	n.d.
Genesis™ 425	M	G	33	M–L	96 (5)	96 (14)	97 (5)	n.d.
Genesis™ Kalkee	T	VG	45	L	100 (5)	102 (17)	94 (5)	n.d.
PBA Monarch	M	F	42	E	101 (5)	101 (17)	97 (5)	n.d.

Yield results are a combined-across-sites analysis using NVT and PBA data from 2011–2015.
Number of trials in brackets (   ).
n.d. = No data. Plant height: T – tall; MT – medium tall; M – medium; MS – medium short.
Lodging resistance: VG – very good; G – good; M – moderate; F – fair; P – poor.
Maturity: E – early; M – medium; L – late.
(Source: Pulse Breeding Australia)

[6] A..Matthews, D McCaffery, L Jenkins (2016), Winter crop variety sowing guide 2016. http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0011/272945/winter-crop-variety-sowing-guide-2016.pdf

Table 3: Chickpea variety ratings for common chickpea diseases in Australia

Variety	Ascochyta blight	Phytophthora root rot[1]	Botrytis grey mould[2]	Virus[3]	Root-lesion nematode (*Pratylenchus thornei*)		Root-lesion nematode (*P. neglectus*)	
					Resistance[4]	Tolerance[4]	Resistance[4]	Tolerance[4]
Desi								
Ambar	R	S	S	–	–	–	–	–
Flipper	MR–MS	MS	S	S	MS	T	MS	–
Howzat	S	MS	MS	S	S	MT	S	MI
Jimbour	S	MS–MR	S	S	S	T	MS	T
Kyabra	S	MS	S	S	VS	–	R	–
Neelam	R	S	S	–	–	–	–	–
PBA Boundary	MR	S	S	S	–	–	–	–
PBA HatTrick	MR	MR	S	S	–	–	–	–
PBA Maiden	MR	S	S	S	–	–	–	–
PBA Slasher	R	S	S	S	–	–	–	–
PBA Striker	MR	S	S	S	–	–	–	–
Yorker	S	MR	S	S	MS	MT	MR	–
Kabuli								
Almaz	MS	VS	S	S	VS	T	MR	–
Genesis™ 090	R	VS	S	S	VS	T	MR	–
Genesis™ 114	MS	VS	S	S	–	–	–	–
Genesis™ 425	R	S	S	S	MS	MI	MR	–
Genesis™ Kalkee	MS	VS	S	S	–	–	–	–
PBA Monarch	MS	VS	S	S	–	–	–	–

R = Resistant, MR = Moderately resistant, MS = Moderately susceptible, S = Susceptible, VS = Very susceptible; T = Tolerant, MI = Moderately intolerant, I = Intolerant, – = No data.

[1] Ratings a compilation of NSW (Tamworth) and Qld (Warwick) data.

[2] The risk of Botrytis grey mould (BGM) damage can be affected by the management of Ascochyta blight (AB); fungicides used to control Ascochyta can also control Botrytis. Note that if BGM risk is high, then a fungicide with greater efficacy for BGM than for AB might also be needed. BGM screening is conducted in a controlled environment and rating is independent of plant architecture.

[3] Virus ratings could change with different virus species predominating in different areas.

[4] Resistance measures the plant's ability to resist disease. Tolerance measures the plant's ability to yield at a given disease level. Tolerant varieties, while potentially yielding well, are unlikely to reduce nematode numbers for following crops. Data supplied by John Thompson, DAF Qld, Toowoomba.

(Source: Pulse Breeding Australia)

An increasing number of pulse variety options exist. Careful variety selection through knowing the agronomic, disease and marketing strengths and weakness of each variety is required to maximise pulse production and returns.

To achieve maximum returns, best agronomic practice needs to be employed according to the variety. These practices include careful paddock selection, planting of high quality seed, and suitable crop protection measures, including weed, disease and insect management, followed by careful harvest, handling and storage practices.

Consideration of market access and options, even prior to crop establishment, can also have a significant impact on the crop's value and profitability. [7]

[7] 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

2.3.2 Desi chickpea variety regional yields

Comparative yield results for Desi chickpea varieties over 5 years are presented for central Queensland and north-eastern Australia (Table 4). Table 5 shows disease resistance ratings and yield loss over 4 years in north-eastern Australia.

Table 4: Comparative yield of desi chickpeas

Long-term yield of desi chickpea, % of PBA PistolA in central Queensland and % of PBA HatTrickA in northeastern Australia (2011–2015)						Yield of desi chickpea, % of PBA HatTrickA, in north-eastern Australia in 2010, a wet winter and spring conducive to AB and BGM					
Variety	Central QLD	South Western QLD	South Eastern QLD	North Western QLD	North Eastern QLD	Tamworth	North Star	Edgeroi	Moree	Bellata	Bullarah
PBA Pistol(D) (t/ha)	2.51	-	-	-	-	-	-	-	-	-	-
PBA HatTrick(D) (t/ha)	2.33	1.9	2.81	1.55	1.94	3.51	1.67	2.26	2.67	1.89	1.82
PBA Seamer(D)	99	104	101	102	101	108	119	137	112	142	118
PBA Boundary(D)	97	104	104	103	102	110	104	105	108	109	93
PBA HatTrick(D)	93	100	100	100	100	100	100	100	100	100	100
PBA Pistol(D)	100	-	-	-	-	-	-	-	-	-	-
Jimbour(D)	94	102	102	101	101	105	56	10	97	96	60
Kyabra(D)	101	107	107	109	106	110	46	6	97	111	60
Moti(D)	101	-	-	-	-	-	-	-	-	-	-

Source: Trial results from Pulse Breeding Australia (PBA) and National Variety Trials (NVT) programs. This report presents NVT "Production Value" MET data on a regional mean basis. This reduces the accuracy and reliability of the results. For detailed PV data, please use the NVT Yield App or Excel Reporting tools available on the NVT website.

Table 5: Disease resistance rating and yield loss of desi chickpea in north-eastern Australia

Variety	Ascochyta blight (AB)[1]					Phytophthora root rot (PRR)[2]				
	Resistance rating	Yield (t/ha)[3]		% Yield loss		Resistance rating	Yield (t/ha)[3]		% Yield loss	
		2014	2015	2014	2015		2014	2015	2014	2015
PBA Seamer(D)	R	2.13	1.57	2	15	MR	1.79	0.37	45	87
PBA Boundary(D)	MR	2.08	1.23	11	30	S	0.73	0.17	74	94
PBA HatTrick(D)	MR	1.76	0.42	23	76	MR	1.98	0.81	33	68
PBA Pistol(D)	S		Not tested			S		Not tested		
Kyabra(D)	S	0.00	0.00	0.00	0.00	MS		Not tested		
Moti(D)	S		Not tested			MS		Not tested		
Yorker(D)	MS		Not tested			2.69	0.57		33	79
PBA Seamer(D) disease free		2.18	1.85				3.23	2.76		

Source: NSW DPI and DAF Pulse pathology and breeding teams

1 Ascochyta blight yield loss trial, Tamworth 2014 & 2015, NSW DPI

2 Phytophthora root rot yield loss trial, Warwick 2014 & 2015, NSW DPI & DAF

3 Yields are in the presence of high disease with no fungicide applications

2.3.3 Desi chickpea variety agronomic traits

Agronomic trait for Desi chickpea varieties are presented for central Queensland (Table 67) and north-eastern Australia (Table 7).

Table 6: Agronomic traits of Desi chickpea in central Queensland

R, Resistant; S, susceptible; M, moderately

Variety	Days to flowering	Plant height (cm)	Lowest pod height (cm)	Lodging Score	Disease resistance rating		
					Ascochyta blight		Phytophthora root rot
					Foliage/ stem	Pods	
Jimbour	65	71	37	4.3	S	S	MS/MR
Kyabra	65	73	37	3.4	S	S	MS
Moti	59	74	37	4.3	S	S	MS
PBA Pistol	58	81	39	2.8	S	S	MS

Source: Pulse Breeding Australia 2005–09.

Table 7: Agronomic traits of desi chickpea in north-eastern Australia

Variety	Flowering (score/days)#		Maturity (score)#		Plant height (cm)		Lowest pod height (cm)		Lodging resistance	Lodging score##	
	Region 2 & 3[1]	Region 1[2]	Region 2 & 3[1]	Region 1[2]	Region 2 & 3[1]	Region 1[2]	Region 2 & 31	Region 1[2]	Region 1[2] 2 & 3[1]	Region 2 & 3[1]	Region 1[2]
PBA Seamer	E-M (4.4)	E-M (67)	M (5.1)	E-M (4.1)	55.2	60.5	30.7	33.0	Good	1.7	2.8
PBA Boundary	M-L (5.9)	M (69)	M (5.3)	M (5.3)	57.8	63.6	35.1	35.4	Mod	2.3	4.5
PBA HatTrick	M (5.0)	E-M (67)	M (5.1)	M (5.0)	56.2	60.7	32.8	33.3	Mod	2.4	4.9
Jimbour	M (4.9)	-	M (4.9)	-	55.3	-	32.6	-	Good	1.8	-
Kyabra	E-M (4.7)	E-M (68)	M (4.9)	E-M (4.5)	55.6	60.9	33.0	34.5	Good	1.7	3.2
PBA Pistol	-	E (65)	-	E (3.5)	-	66.7	-	35.0	-	-	2.8
Moti	-	E-M (68)	-	E-M (4.1)	-	60.2	-	34.4	-	-	2.8

 # Flower & Maturity score, 1 = very early, 9 = very late (E=early, M=mid, L=late)

##Lodging score, 1 = fully erect, 9 = flat on ground

1 Data collected from sites in southern QLD and northern NSW (2011–2015)

2 Data collected from sites in central QLD (2012–2015)

2.3.4 Desi chickpea grain quality

Grain quality (seed weight and dhal quality) for Desi chickpea varieties in northern NSW and southern and central Queensland are presented in Table 8. Availability of seed, EPRs and restrictions are in Table 9.

More information

http://www.daf.qld.gov.au/plants/field-crops-and-pastures/broadacre-field-crops/chickpeas/varieties

http://www.grdc.com.au/Resources/Factsheets/2013/05/Chickpea-disease-management

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Chickpea-varietal-purity-and-implications-for-disease-management

Table 8: Grain quality of Desi chickpea in north-eastern Australia and central Queensland

Variety	Seed weight (g/100)		Dhal yield (%)	
	Regions 2 and 3	Region 1	Regions 2 and 3	Region 1
Jimbour⚛	19.8	22.0	64.2	66.7
Kyabra⚛	25.3	25.3	64.9	63.9
Moti⚛	–	22.2	–	63.8
PBA Boundary⚛	19.5	–	64.7	–
PBA HatTrick⚛	20.1	19.5	65.1	66.9
PBA Pistol⚛	–	24.7	–	67.2
Yorker⚛	21.2	–	62.9	–

Regions 2 and 3, northern NSW and southern Queensland; Region 1, central Queensland
Source: Pulse Breeding Australia.

Table 9: Desi chickpea variety seed availability

Variety	PBR	Licensee or agency	Commercial partner	Seed supplying agents	Telephone	End point royalty ($/t, incl GST), market restriction
Jimbour⚛	PBR	DAFF Qld	Mount Tyson Seeds	Retail outlets	07 4693 7166	Seed royalty, none
Kyabra⚛	PBR	DAFWA	Seedmark	Seedmark	1800 112 400	Seed royalty, none
Moti⚛	PBR	DAFWA	Seednet	Seednet	1800 054 433	$2.75, none, central Qld only
PBA Boundary⚛	PBR	PBA	Seednet	Seednet	1800 054 433	$4.40, none
PBA HatTrick⚛	PBR	PBA	Seednet	Seednet	1800 054 433	$4.40, none
PBA Pistol⚛	PBR	PBA	Seednet	Seednet	1800 054 433	$4.40, none, central Qld only
Yorker⚛	PBR	NSW DPI	Seednet	Seednet	1800 054 433	$3.30, none

[A]Australian Agricultural Crop Technologies. [8]

2.4 Disease management and varietal resistance

2.4.1 Ascochyta blight

Ascochyta blight (caused by *Phoma rabiei*) is a serious disease of chickpeas in Australia (Figure 5). It is now endemic in all growing regions including central Queensland.

Considerable advances have been made with recent varietal releases in terms of Ascochyta blight resistance. However, several other diseases need to be considered for individual situation risks when selecting varieties and paddocks. The most important of these risks is the soil-borne disease Phytophthora root rot.

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

Figure 5: Ascochyta blight. (Photo: Gordon Cumming, Pulse Breeding Australia)

Table 10 shows the relative yield responses of varieties to Ascochyta blight and Phytophthora root rot, in situations of high disease pressure. The yield advantages of varieties such as PBA Boundary⍉ and Kyabra⍉ can be quickly lost if they are exposed to diseases to which they are susceptible.

Growers need to determine the variety's disease risk profile as they may be better served by selecting the variety with the greatest varietal resistance(s) to the expected disease pressures, even if it is lower yielding in disease-free situations.

These decisions should be made in conjunction with an understanding of what management options are available. [9]

Table 10: Disease resistance rating and yield loss of Desi chickpea in north-eastern Australia

Variety	Ascochyta blight[A]					Phytophthora root rot[B]		
	Resistance rating	Yield (t/ha) 2009	Yield (t/ha) 2010	% Yield loss 2009	% Yield loss 2010	Resistance rating	Yield (t/ha)	% Yield loss[C]
Jimbour⍉	S	0.44	0.00	77	100	MS/MR	2.70	66
Kyabra⍉	S	–	0.00	0	100	MS	2.83	78
PBA Boundary⍉	MR	1.84	2.32	4	4	S	2.58	85
PBA HatTrick⍉	MR	1.71	1.71	8	34	MR	2.56	64
Yorker⍉	MS	1.80	–	5	–	MR	2.52	35

Yields are in absence of infection. R, resistant; S, susceptible; M, moderately

Source: NSW DPI and DAFF Qld Plant pathology teams

[A]Ascochyta blight yield loss trial, Tamworth 2009 and 2010, NSW-DPI.

[B]Phytophthora root rot yield loss trial, Warwick 2012, DAFF Qld and NSW DPI.

[C]% Yield loss due to inoculation with Phytophthora root rot.

[9] 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

2.4.2 Phytophthora root rot

Phytophthora root rot is a soil-borne disease that can establish permanently in some paddocks. The year 2010 was particularly conducive to Phytophthora root rot because damage is greatest in seasons with above-average rainfall. However, only a single saturating rain event is needed for infection, as was seen in 2012.

Management options for Phytophthora root rot

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 blight. Thus, Phytophthora root rot can be managed only by pre-sowing decisions and assessing risks for individual paddocks.

Development of the disease requires the pathogen in the soil, and a period of soil saturation. Losses in a *Phytophthora*-infested paddock may be minor if soil saturation does not occur.

The most effective control strategy is to avoid sowing chickpeas in high-risk paddocks, which are those with a history of:

- Phytophthora root rot noted in previous chickpea or lucerne crops
- lucerne or annual or perennial medics
- waterlogging or flooding

If chickpea is to be sown in high-risk paddocks, then growing a variety with the highest level of resistance (Table 11) may reduce losses from Phytophthora root rot. This is particularly the case in medium-risk situations, where medic, chickpea or lucerne crops have been grown in the past 5–6 years. However, the level of protection offered by varietal resistance remains low. [10]

More information

https://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/02/Rootlesion-nematode-management-the-cost-of-getting-it-wrong

https://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2010/05/Chickpeas-In-2010-PBA-Hattrick-Performance-And-Management

2.4.3 Nematodes

Nematodes are minute, worm-like parasites that attack the root system of susceptible crops. They are usually <1 mm long. The most obvious symptom of nematode damage is patchy, uneven crop development. Approximately 70% of the wheat-based farming country in southern Queensland is infested to varying degrees with root-lesion nematodes. In paddocks with moderate nematode numbers, relatively susceptible varieties of chickpeas such as Amethyst or Sona can suffer a yield loss of up to 15–30%, whereas more tolerant varieties such as Jimbour might suffer a yield loss of 5–10% (Table 11). Soil test levels above 2000 nematodes/kg soil are likely to affect chickpea yields (based on 0–30 cm soil sampling).

Nematode numbers will also build up on the root system of chickpeas. The extent to which root-lesion nematodes numbers build up on the crop root system (resistance or susceptibility) may impact on subsequent crops in the rotation. Nematode build-up on chickpeas is similar to that on other susceptible hosts (barley, maize, and triticale), but much less (<50%) than on susceptible wheat varieties.

[10] 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

Table 11: Chickpea variety root-lesion nematode ratings 2008

Variety	*P. thornei* Resistance	*P. thornei* Tolerance	*P. neglectus* Resistance	*P. neglectus* Tolerance
Desi types				
Amethyst	S	MT	MS	T
Flipper⚊	MS	T	MS	–
Howzat	S	MT	S	MI
Jimbour⚊	S	T	MS	T
Kyabra⚊	VS	–	R	–
Moti⚊	S	T	S	T
Yorker⚊	MS	MT	MR	–
Kabuli types				
Almaz⚊	VS	T	MR	–
Genesis™ 090	VS	T	MR	–
Genesis™ 425	MS	MI	MR	–
Kaniva	MS	MT	MR	I
Macarena	S	–	VS	–
Nafice⚊	VS	MR	R	–

R, resistant; S, susceptible; M, moderately; V, very; T, tolerant; I, intolerant. Resistance measures the plant's ability to resist disease; tolerance measures the plant's ability to yield given disease level; tolerant varieties, while potentially yielding well, are unlikely to reduce nematode numbers for following crops (data supplied by John Thompson, DAFF, Toowoomba)

More information

http://www.nga.org.au/results-and-publications/download/139/project-reports-diseases/multi-trial-summary-1/root-lesion-nematode-crop-variety-impact-on-populations-2011.pdf

Two species of root-lesion nematode, *Pratylenchus neglectus* and *P. thornei,* occur in the cropping regions of northern Australia. Both species cause root damage and yield losses. Root-lesion nematodes have a wide host range, including cereals and grassy weeds, pulses, pasture and forage legumes and oilseeds.

Chickpeas are susceptible to both *Pratylenchus thornei* and *P. neglectus*. Mungbean, faba bean and soybean are susceptible to *P. thornei* but resistant to *P. neglectus*.

Chickpeas will result in increased levels of *Pratylenchus* after the crop, although some varietal differences are apparent. The newer varieties PBA HatTrick⚊ and PBA Boundary ⚊ have consistently shown lower *Pratylenchus* levels than Jimbour⚊ and Kyabra⚊ (Figure 6). [11]

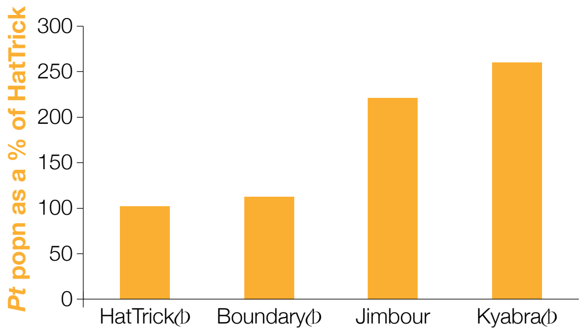


Figure 6: All varieties evaluated in 9 trials 2011-13 (DAFF, NSW DPI, NVT and NGA)

2.4.4 Viral diseases

At least 14 viruses cause significant losses in chickpea, and currently all northern chickpea varieties are considered susceptible to them.

[11] 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

The occurrence of virus in chickpea changes dramatically from season to season and location.

Control measures for viruses in chickpea are not adequate at present. Application of seed and foliar insecticides, aimed at preventing feeding by aphids, has failed to prevent infection by viruses in field experiments.

Best agronomic management can help to reduce damage by viruses and includes:

- retention of cereal stubble to deter aphids;
- sowing at recommended times to avoid autumn aphid flights;
- sowing at recommended seed densities to achieve early closure of the crop canopy (closed canopies deter aphids) [12]
- effective weed control (particularly marshmallow)

2.5 Future breeding directions

More information

Pulse Australia (2015), Chickpea: High Quality Seed

GRDC (2013), Clean seed and care the recipe for chickpea success

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.

The most likely next release for southern Queensland and northern New South Wales is the coded line CICA0912, which combines the yield potential of the current commercial lines as well as the best Ascochyta blight and Phytophthora root rot that is currently available in a single variety.

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 [13]

2.6 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 being infected with seed-borne pathogens:

- All seed should be tested for quality including germination and vigour.
- If grower-retained seed is of low quality, consider purchasing registered or certified seed from a commercial supplier and always ask for a copy of the germination report.
- Regardless of the source, 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).

Good establishment through correct plant density and good seedling vigour is important to maximise yields of pulse crops (Figure 7). A targeted density can only be achieved by having quality seed with good vigour and a known germination percentage to accurately

12 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

13 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

More information

http://grdc.com.au/Media-Centre/Ground-Cover-Supplements/Ground-Cover-issue-94-SepOct-2011-Crop-Protection-Supplement/Chickpea-seed-research-to-improve-crop-establishment

http://agvantagecommodities.com.au/nsw-dpi-chickpea-seed-testing-service-available-at-tamworth/

http://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2012/04/Chickpea-seed-tests-from-2010-harvest-explain-establishment-problems-in-2011-crops

http://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2012/04/Chickpea-seed-treatment-improves-crop-establishment-and-increases-yields-2011-trials-using-seed-from-2010-crops

calculate seeding rates. A slight variation in seed size due to seasonal conditions or an incorrect germination percentage can make a huge difference in the calculated seeding rates required to achieve a satisfactory target plant density.

Many seed buyers are unaware that minimum germination requirement for certified pulse seed is 70%, which is far less than the 90% or greater often obtained in pulse seed. Many believe that this minimum should be raised to 80%, as not all growers or retailers request seed test results of certified pulse seed. Test results must be made available under the Seeds Act and Australian Seeds Federation guidelines, so ask for it.

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.

Seed with low germination rates and poor seedling vigour can result in sparse establishment and a weak crop, which then becomes more vulnerable to viruses, fungal disease infection and insect attack, and is less competitive with weeds. Inevitably, this will result in significantly lower yields. The crop may also have variable maturity rates, making it difficult to manage.

Some pulses, such as field pea, may have low germination rates of 'normal seedlings' because they initially have a high percentage of dormant seed. The large size or fragile nature of pulse seed, particularly lupin, 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.

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. [14]

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

Figure 7: Good establishment through correct plant density and good seedling vigour is important to maximise yields of pulse crops.

2.6.1 Grower-retained planting 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 should be professionally graded to remove unviable seeds and weed seeds, and treated with a thiram-based seed treatment.

Seed-borne diseases can lower germination levels, and testing for presence in seed can be conducted by specialist laboratories for 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.

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.

The only way to accurately know the seed's germination rate, vigour and disease level is to have it tested. [15]

More information

L Jenkins, K Moore, G Cumming (2015). Chickpea: high quality seed

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

More information

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Chickpea-varietal-purity-and-implications-for-disease-management

GRDC (2014), Grain Storage Fact Sheet: Storing Pulses

2.6.2 Safe storage of seed

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. [16]

See GRDC GrowNotes (Chickpeas) Section 13, Storage.

Retained seed needs to be stored safely to ensure its quality is maintained. Safest storing conditions for pulses are at 20°C and at 12.5% moisture content (Table 12).

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 moisture and temperature increases longevity of the seed, although storage at very low moisture contents (<10%) may render chickpea more vulnerable to mechanical damage during subsequent handling as the seed pulls away from the seed-coat.

Table 12: Effect of moisture content and temperature on storage life of chickpea seed

Storage moisture	Storage temperature	Longevity of seed
12%	20°C	>200 days
	30°C	500–650 days
	40°C	110–130 days
15%	20°C	700–850 days
	30°C	180–210 days
	40°C	30–50 days

Note: Most sowing seed will need to be stored for a ≥180 days

Source: Ellis et al. (1982).

Storage at moisture levels >13% under Australian conditions is not recommended. Reducing temperature in storage facilities is the easiest method of increasing seed longevity. Not only will it increase the viable lifespan of the seed, it will also slow the rate at which insect pests multiply in the grain.

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. [17]

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

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

GRDC GRAINS RESEARCH & DEVELOPMENT CORPORATION

More information

http://storedgrain.com.au/wp-content/uploads/2013/06/chickpea_harvest_storage.pdf

For more information, see GRDC GrowNotes (Chickpeas) Section 13, Storage.

2.6.3 Handling bulk 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. [18]

More information

GRDC (2015) Chickpea seed testing

2.7 Seed testing

2.7.1 Germination testing

Germination tests can be conducted by a simple home test, or ideally by sending a representative sample to seed-testing laboratories for germination and vigour tests. For beans, chickpea, lupins, peas and vetch, the sample size required is 1 kg for each 25 t of seed. For lentils, take 1 kg for each 10 t of seed.

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. Mix subsamples thoroughly and then take a composite sample of 1 kg. 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.

2.7.2 Vigour testing

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. Otherwise it is wise to do your own test by sowing seeds into a soil tray that is kept cold (<20°C).

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.

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

2.7.3 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 normal germination after ageing (vigorous seedlings).

2.7.4 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.

2.7.5 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:

- 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) 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.

2.7.6 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.

2.7.7 Other

Another example of a vigour test used by some Australian laboratories is germination testing at 7°C for 12–20 days in the dark and under low moisture conditions. If seed vigour is acceptable, then this germination result should be within 10% of the regular germination test.

2.7.8 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

More information

https://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2012/04/Chickpea-seed-tests-from-2010-harvest-explain-establishment-problems-in-2011-crops

of a seed sample can be conducted if requested, including the amount of weed seed contamination.

2.7.9 Disease testing

Seed-borne diseases 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.

For a disease test, 1 kg of seed is required, except for anthracnose where 2 kg is needed.

2.7.10 Major pathogens identified in seed tests

The NSW DPI plant pathology team offers a free seed-testing service as part of a GRDC-funded Integrated Disease Management project (Figure 8).

Seed samples will be tested for germination, vigour and pathology (i.e. Botrytis grey mould and Ascochyta blight infections) with the results forwarded to the supplier of the sample. Major pathogens identified in these tests are listed in Table 13.

If you wish to have samples tested, please forward a 1-kg sample of harvested chickpea seed to:
Dr Kevin Moore
C/- Gail Chiplin
Tamworth Agricultural Institute
4 Marsden Park Rd
Calala, NSW 2340

Dr Jenny Wood, NSW DPI Tamworth, is also testing seed as part of a GRDC-funded project to eliminate grain defects. The 1-kg sample can be analysed as part of this project and undergo testing by the pathology team as described above.

For more information, contact:
Dr Jenny Wood on jenny.wood@dpi.nsw.gov.au or phone 02 6763 1157.

Figure 8: Testing seed before sowing will identify the presence of disease and allow steps to be taken to reduce the disease risk. NSW DPI Tamworth offers this service. (Photo: Rachel Bowman, Seedbed Media)

Table 13: Major chickpea pathogens identified in seed tests

Check the test number against the laboratories that do seed tests

Test no.	Pathogen	Disease
1	*Ascochyta rabiei (Phoma rabiei)*	Ascochyta blight
2	*Botrytis cinerea*	Botrytis grey mould
3	*Cucumber mosaic virus* (CMV)	CMV

Refer to *GRDC GrowNotes (Chickpeas) Section 9, Disease management.*

Listed below are laboratories that will test for some or all of the diseases in Table 14.

SARDI Field Crops Pathology
GPO Box 379, Adelaide, SA 5001
Telephone (08) 8303 9384 Facsimile (08) 8303 9393
Tests: 1, 2
Web: http://www.sardi.sa.gov.au/crops/new_variety_agronomy/services/seed_testing_and_sampling

Asure Quality Australia
3-5 Lillee Cresent, PO Box 1335, Tullamarine, Vic. 3043
Telephone (03) 8318 9000 Facsimile (03) 8318 9001
Tests: 1–3
Web: www.asurequality.com

2.7.11 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 (e.g. Jimbour) are susceptible to Ascochyta blight and, in seasons conducive to disease, require intensive management with foliar fungicides. Most cultivars released in 2005 and later, such as PBA HatTrick, have improved Ascochyta blight resistance and require fewer fungicide sprays.

Since 2011, several chickpea crops in the GRDC northern region have shown inconsistencies in their reactions to Ascochyta blight. In all cases, the variety was named as PBA HatTrick and the seed was grower-retained. PBA HatTrick, released in 2009, is rated moderately resistant to Ascochyta blight but the level of disease in these crops was more typical of varieties rated as susceptible.

Possible explanations for these unexpected higher levels of disease include:

- a change in the pathogenicity of *Phoma rabiei* (i.e. breakdown of varietal resistance)
- authenticity and/or purity of the variety (i.e. mix up in seed source or contamination).

A comprehensive study of the Australian population of *Phoma rabiei* found low genetic diversity of isolates and little evidence for widespread changes in pathogenicity. Simpfendorfer *et al.* (2013) showed that varietal contamination caused the higher than expected levels of stripe rust in the moderately resistant bread wheat variety Sunvale. This posed the question: could contamination or a mix-up in source of planting seed account for the observed differences in Ascochyta blight levels in PBA HatTrick crops grown from grower-retained seed? It also raised the larger issue of maintaining genetic purity in Australian chickpea varieties after their release.

Key points:

- 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 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.

Figure 9: As the issue of seed purity increases, growers should treat crops from suspect seed as a susceptible variety. (Photo: Rachel Bowman, Seedbed Media)

How widespread is the purity poblem?

It has not yet been established, but NSW DPI 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 (Figure 9).

In 2013, on a property near Moree, three paddocks had been planted with seed from three different sources, all grower-retained and all believed to be PBA HatTrick⚘. When inspected on 8 and 9 August 2013, it was obvious that one of the paddocks was different from the other two and was clearly not PBA HatTrick⚘ (possibly Howzat).

A similar situation was observed, again in 2013, on another north-western NSW property where the grower had sown one half of a paddock with grower-retained seed and the other half with a different source of grower-retained seed. The seed from the two sources was believed to be PBA HatTrick⚘, but when inspected, it was obvious that they were not the same variety and again one was not PBA HatTrick⚘ (possibly Yorker⚘).

Does it matter if a chickpea crop is a mixture of varieties?

Why is it important to know which variety you are growing and the level of contamination, if any? 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 grower's legal rights (e.g. if seed you purchased is not what you paid for)
- assessing compliance with plant breeder's 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)

More information

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Chickpea-varietal-purity-and-implications-for-disease-management

Cost of Ascochyta blight management—an example of a consequence of varietal impurity

In a season that is conducive to chickpea Ascochyta, Tamworth-based NSW DPI research has shown that a crop of pure PBA HatTrick⑴ will require two foliar fungicide sprays totaling $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. [19]

2.7.12 Performing your own germination and vigour test

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.

For your own germination test, use a flat, shallow seeding tray about 5 cm deep. Place a sheet of newspaper on the base to cover drainage holes. Use clean sand, potting mix or a freely draining soil. Testing must be at a temperature of <20°C, so testing indoors may be required. Randomly count out 100 seeds per test, but do not discard any damaged seeds.

If the tray has been filled with soil, sow 10 rows of 10 seeds in a grid at the correct seeding depth. Do this by placing the seed on the levelled soil surface and gently pushing each in with a pencil marked to the required depth. Cover seed holes with a little more soil and water gently (Figure 10).

Alternatively, place a layer of moist soil in the tray and level it to the depth of sowing that will be required. Place the seeds as 10 rows of 10 seeds in a grid on the seedbed formed. Then uniformly fill the tray with soil to the required depth of seed coverage (i.e. seeding depth). Ensure that the soil surface is uniformly levelled, and water gently if required.

During the test, keep the soil moist, but not wet. Overwatering will result in fungal growth and possible rotting. After 7–14 days, the majority of viable seeds will have emerged. Count only normal, healthy seedlings. The number of normal and vigorous seedlings you count will be the germination percentage.

[19] 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

This germination test is in part a form of inbuilt vigour testing because it is done in soil. To further establish vigour under more adverse conditions, a second germination test done under colder or wetter conditions could be used as a comparison with the normal germination test, done at the same time.

Figure 10: Doing your own germination test. (Photo: E. Leonard, AgriKnowHow)

2.7.13 Safe rates of fertiliser sown with the seed

All pulses can be affected by fertiliser toxicity. Lupins are especially susceptible. Higher rates of phosphorus (P) fertiliser can be toxic to lupin 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, such as in northern NSW with rates of P at 50 kg/ha of DAP on 1-m rows with 4 cm of in-row disturbance.

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 often preferred for lupins, or else broadcasting and incorporating, drilling pre-seeding or splitting fertiliser applications so that lower rates or no P is in contact with the seed. [20]

More information

http://grdc.com.au/Resources/Factsheets/2011/05/Fertiliser-Toxicity

http://www.grdc.com.au/uploads/documents/4 Nutrition.pdf

[20] GRDC (2008) Grain Legume Handbook update 7 Feb 2008. Grain Legume Handbook Committee, supported by the Grains Research and Development Corporation (GRDC).

SECTION 3

Planting

More information

www.grdc.com.au/Resources/Publications/2013/09/Inoculating-legumes-back-pocket-guide

QDAF (2015) Planting chickpeas, Inoculation and nodulation

GRDC (2012), Inoculating legumes: a practical guide

3.1 Inoculation

Pulses have the ability to fix their own nitrogen (N) from the air via nodules on their roots if specific N-fixing bacteria (rhizobia) are available. [1]

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. 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.

Sampling of commercial paddocks in Queensland and New South Wales (NSW) has revealed that poor inoculation practices result in ineffective nodulation. The most effective method to ensure nodulation with the applied strain of inoculum is delivery of the highest possible concentration of live cultures onto the seed and sowing into good moisture as quickly as possible.

The most common method of inoculating chickpea is to coat the seed with a slurry of peat-based inoculum immediately before planting (Figure 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]

Figure 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.)

3.1.1 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.

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

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

Figure 2: Well-nodulated chickpea plants. (Photo: G. Cumming, Pulse Australia)

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 (Figure 2).

More information

http://www.grdc.com.au/GRDC-FS-NFixation-Chickpeas

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.

The Group N bacteria are regarded as an 'aggressive nodulator'. This effectively means that nodulation will be successful in meeting the crop's N requirements, provided inoculants are handled and stored in a manner that will ensure bacterial survival, and growers adopt effective inoculation practices.

Inoculated seed is best planted into moisture within 12 h of treatment, as fungicide seed dressings can affect survival of the bacteria.

Group N rhizobia are extremely sensitive to the level of available nitrate-N in the soil. Although high levels of nitrate-N have no significant effect on both the initial formation and number of nodules, they do markedly reduce both nodule size and activity.

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 colour inside (Figure 3).

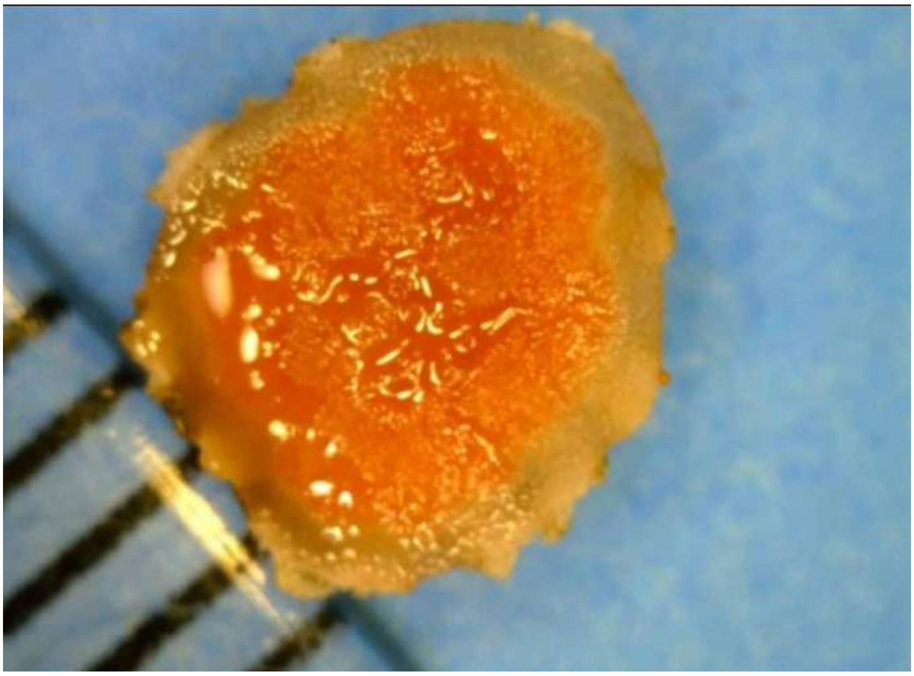


Figure 3: Active nodules have a rusty red or pink centre. (Photo: G. Cumming, Pulse Australia)

Growing chickpeas on long fallows or in a situation with high residual N will substantially reduce N fixation (Figure 4).

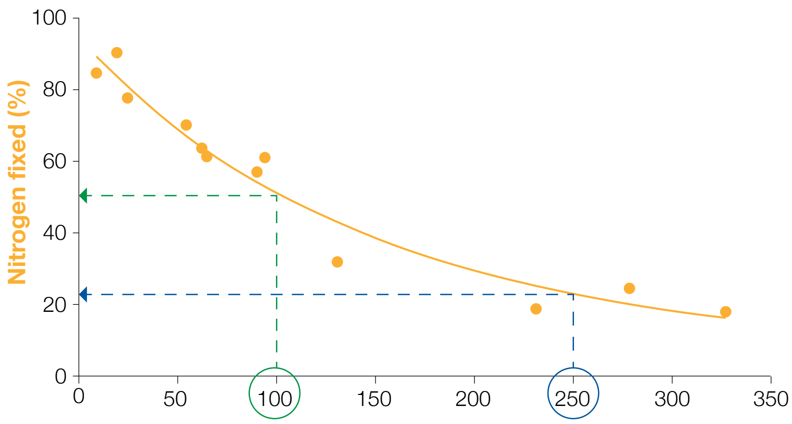


Figure 4: Effect of soil NO3-N at planting (kg/ha, 0–120 cm) on per cent nitrogen fixed in chickpea (cv. Reselected Tyson) shoots at 130 days after planting for various levels of soil NO3-N at crop establishment. For fitted curve, Y = 7.05 + 88.45e-0.0070X, R2 = 0.95. (Source: JA Doughton et al. 1993).

Even growing chickpeas on summer fallows after wheat (6-month fallow) will delay the onset of N fixation due to the mineralisation of 30–50 kg N/ha in the fallow period (Table 1). This nitrate-N, coupled with further in-crop mineralisation (15–20 kg/ha), provides a total soil supply of 45–70 kg N/ha, which is sufficient to grow a 1 t/ha chickpea crop. Yields above this level are completely dependent on N fixation (and effective nodulation practices).

If poor nodulation and N deficiency are to be eliminated as a major constraint to chickpea production, growers need to pay much greater attention to inoculation practices, and to treatment and handling of inoculant materials. [3]

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

Table 1: Nitrogen amounts fixed in chickpea crops

	Chickpea grain yield (t/ha)	Nitrogen fixation (kg/ha)
Double-cropped from sorghum	2.4	103
On long fallow (18 months)	2.4	27

Source: JA Doughton et al. 1993.

3.1.2 Management impacts on N fixation

- Changes to agronomy can change N fixation in grain legumes.
- In general, increasing row spacing may decrease amount of N fixed by legumes.
- Varieties can differ significantly in amount of N fixation and this is related to biomass.
- High soil nitrate levels can reduce legume nodulation and N fixation by rhizobia.

Average amounts of N fixed annually by crop and pasture legumes are about 110 kg N/ha (ranging from close to zero to >400 kg N/ha). The actual amount fixed depends on the species of legume grown, the site and the seasonal conditions as well as agronomic management of the crop or pasture. The legume crop uses this N for its own growth and may fix significantly more than needed, leaving a positive N balance in the soil for proceeding crops.

The amount of N fixed by a legume increases as legume biomass increases but is reduced by high levels of soil nitrate. In general, legume reliance on N fixation is high when soil nitrate levels are <50 kg N/ha in the top 1 m of soil. Above 200 kg N/ha, N fixation is generally close to zero. The fixed N is used for the growth of the legume itself (saving fertiliser application of the legume crop) as well as potentially leaving residual N for the following cereal or oilseed crop and providing a break from cereal stubble and soil-borne diseases.

Work by Doughton *et al*. (1993) clearly demonstrated the impact of increasing soil nitrate levels on N fixation of chickpeas (Figure 4), with no yield advantage being gained by applying N. Moreover, chickpea provided a positive soil N balance when fixation rates were high and a negative balance at low fixation rates. [4]

Individual trial results from a site near Bellata, NSW, in 2012, with a soil nitrate level of 69 kg N/ha at 0–90 cm, showed no yield advantage from applying N (Figure 5).

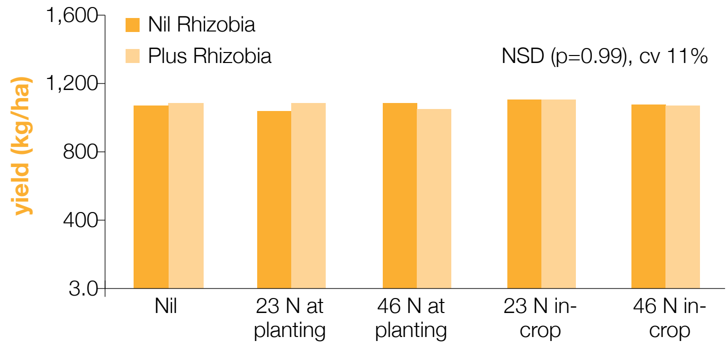


Figure 5: Yield of chickpea at high soil nitrate levels. No significant yield gain was obtained by applying N at planting or in-crop, or by adding rhizobia. (Source: NGA Project Reports 2012)

More information

http://www.nga.org.au/results-and-publications/download/223/project-reports-general/complete-summary/nitrogen-application-in-chickpeas-2012-complete-summary.pdf

[4] N Seymour, RCN Rachaputi, R Daniel (2014) Management impacts on N fixation of mungbeans and chickpeas. GRDC Update Papers 4 March 2014, http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Management-impacts-on-N-fixation-of-mungbeans-and-chickpeas

More information

L Drew, R Ballard, M Denton (2011), Inoculation to optimise pulse performance

3.1.3 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 (Figure 6).

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. [5]

The current recommendation is to inoculate chickpea every time it is grown due to poor rhizobium survival on northern region alkaline soils.

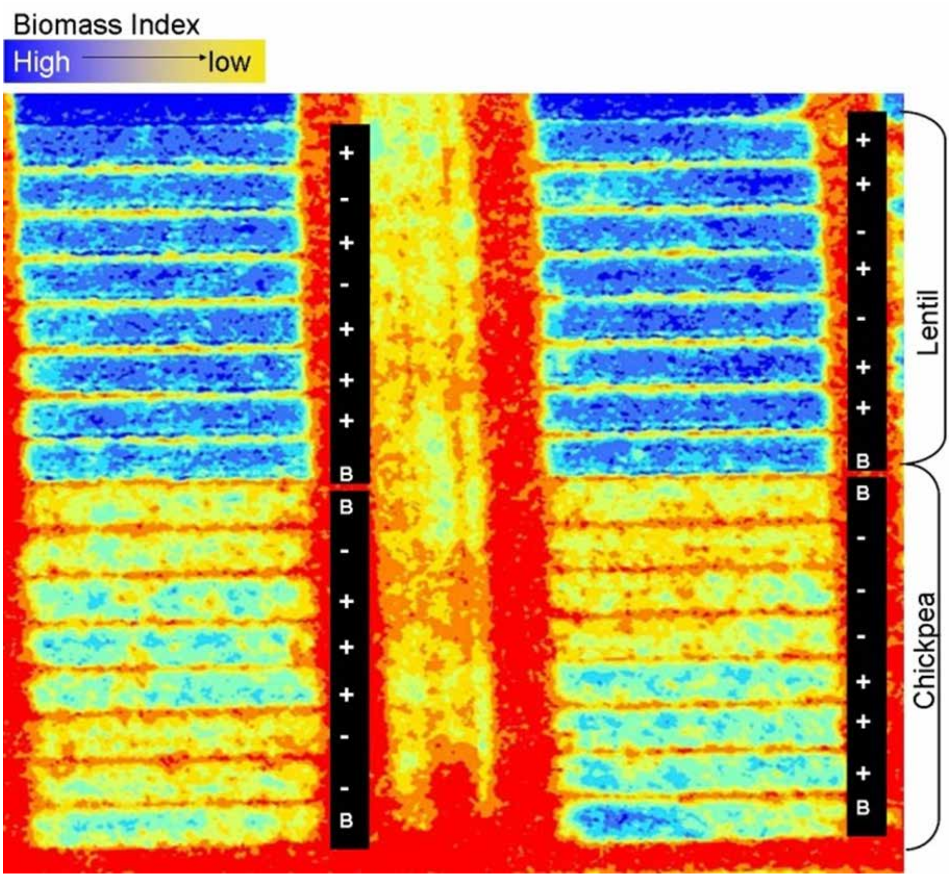


Figure 6: Biomass image of lentil (top) and chickpea (bottom) inoculant plots at Paskeville SA 2009. Blue represents high biomass, yellow represents low biomass and red is bare earth. Note the responsiveness of chickpea but not lentil, plots to inoculation and inoculant type. B, buffer; +, inoculated; –, uninoculated. (Photo: National Rhizobium Program trial by J. Heap, SARDI)

3.1.4 Storing inoculants

For maximum survival, peat inoculant should be stored in a refrigerator at about 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. [6]

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

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

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 h. Sow seed inoculated with peat slurry as soon as possible, but certainly within 12 h, 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 5 h 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. [7]

More information

NSW DPI, Australian Inoculants Research Group

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). [8]

Table 2: ALIRU Quality Assurance rhizobia minimum numbers

Product	Viable rhizobia (no./g)	Rate (/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

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). [9]

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.

9 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[A]	Compatibility with seed-applied fungicide
Peat inoculums	Seed	Low	Some (check label)
Freeze-dried inoculums	Seed or in-furrow (water inject)	Very low	Very low
Granular forms	Seeding furrow or below seed	Yes	Yes
In-furrow water injection	Seeding furrow or below seed	Very low	Yes

[A]Survival will depend on duration of dry conditions and soil pH.

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 (Figure 7)
- 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.

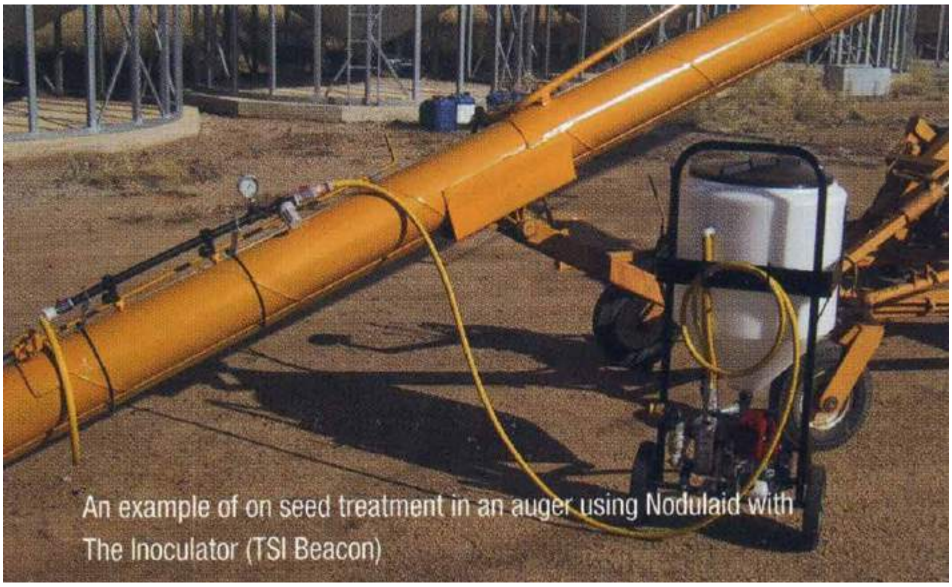


Figure 7: Two nozzles mounted into the top of the auger case 1 m apart.

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 8). Its mixing ability is not as effective as an auger. [10]

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

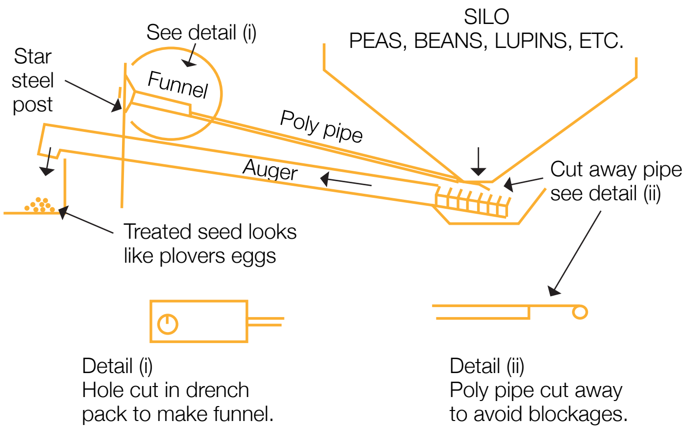


Figure 8: Application of inoculum to seed through a tubulator.

3.1.9 Newer inoculation methods

With new inoculant types and technologies, an appreciation is needed of each type's strengths and limitations. Rhizobial survival becomes more important under difficult circumstances such as prolonged dry soil conditions, when seed treatment of either fungicide or trace elements is used, and depending on soil pH. Much of the survival is associated with the degree of protection the rhizobia has against drying or against adverse conditions. Ease of inoculant application is increasingly important and needs to be accounted for in costing. [11]

3.1.10 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.

Queensland Department of Agriculture, Fisheries and Forestry (QDAFF) trials have consistently shown superior nodulation from water injection of inoculum (Figures 9 and 10). This is likely to be due to the larger numbers of live bacteria being delivered into the soil in close proximity to the seed.

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

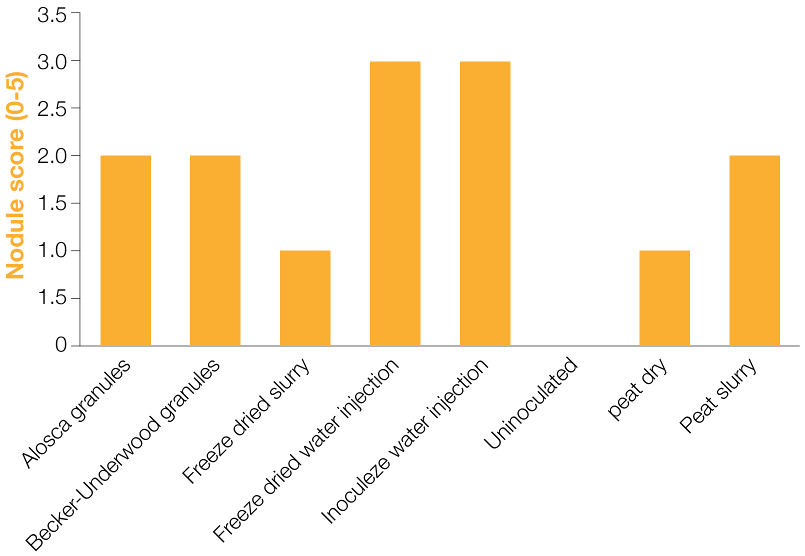


Figure 9: Inoculation trial at Emerald (2005), nodule scores assessed at 4 weeks after planting.

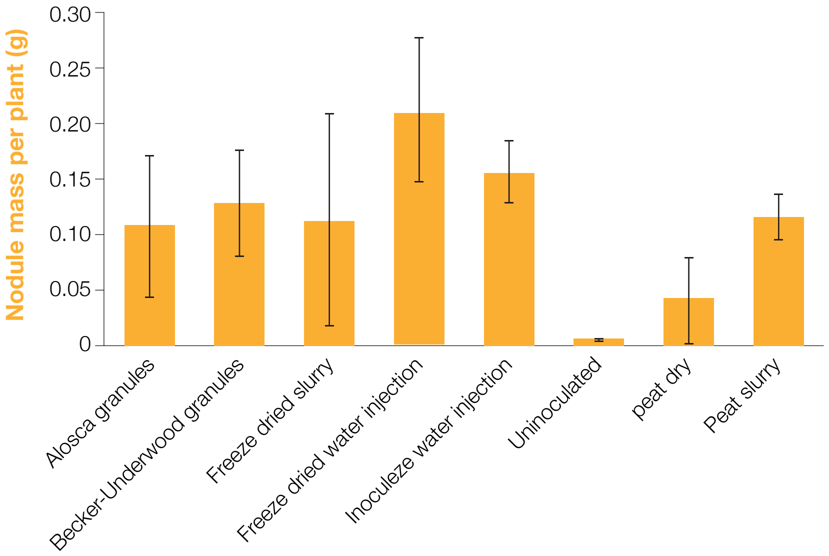


Figure 10: Inoculation trial at Emerald (2005), nodule biomass at flowering.

No significant differences were found in shoot weight (4 weeks after planting) or in final biomass and grain yield. [12]

Figures 11, 12 and 13 depict liquid injection set-ups on seeding equipment, and Figure 14 shows the liquid stream.

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

Figure 11: A seeder set up with Atom Jet narrow points, gang press wheels and liquid injection for either inoculum or trace element application during sowing.

Figure 12: A disc seeder set up with Yetter trash clearing wheels and tubing for liquid injection of either inoculum or trace elements during sowing. Note also the closer to cover the seeding slot and act like a 'press' wheel' from the side.

Figure 13: Tanks mounted on the seeding bar for liquid injection of either rhizobium or trace elements during seeding. Agitation is required. Note the tubes and manifold. Inoculum must be applied under low pressure only. Some machines have their tanks set up as a separate trailed tanker.

Figure 14: In furrow-liquid injection: Note the droplets from liquid injection, which can be used for either inoculating pulses or applying liquid trace elements. (Source: Grain Legume Handbook)

3.1.11 Granular inoculants

Granular inoculants are applied like fertiliser as a solid into the seed furrow, near to the seed or below. They avoid many of the compatibility problems of rhizobia with fertilisers and fungicides. They also eliminate the need to inoculate seed before sowing. Granulars may also be better where dry sowing is practiced or sowing into acidic soils because the rhizobia survive better than on seed (Table 3). A third, small seed box is required to apply granular inoculum (Figure 15). This is because rhizobial survival is jeopardized if the granular inoculum is mixed with fertilizer. If it is mixed with the seed, then distribution of both seed and inoculum is affected, causing either poor and uneven establishment and/or patchy nodulation.

Granules contain fewer rhizobia per gram than peat-based inoculants, so they must be applied at higher application rates. The size, form, uniformity, moisture and rate of application of granules differ between products. Depending on product or row spacing, rates can vary from 2–10 kg/ha to deliver comparable levels of nodulation. [13]

Figure 15: 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)

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

3.1.12 Inoculant 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 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, including those listed in Table 4. Figure 16 presents trial results in terms of nodule score by product type.

Table 4: Inoculant treatments

Inoculant	Description
Nodulaid™	Standard peat slurry applied to seed (PS)
Nodulaid™ Water Inject	Peat slurry injected into seed furrow with water as a carrier (PS WI)
Nodulator™	Attapulgite clay granules mixed with seed in furrow (ACG)
EasyRhiz®	Freeze-dried rhizobia mixed into slurry and applied direct to seed (FD)
EasyRhiz® Water Inject	Freeze-dried rhizobia slurry injected into seed furrow with water as a carrier (FD WI)
Alosca® granules	Bentonite clay granules mixed with seed in furrow (BCG)
N-Prove® granules (available now only as TagTeam®)	Peat granules mixed with seed in furrow (PG)

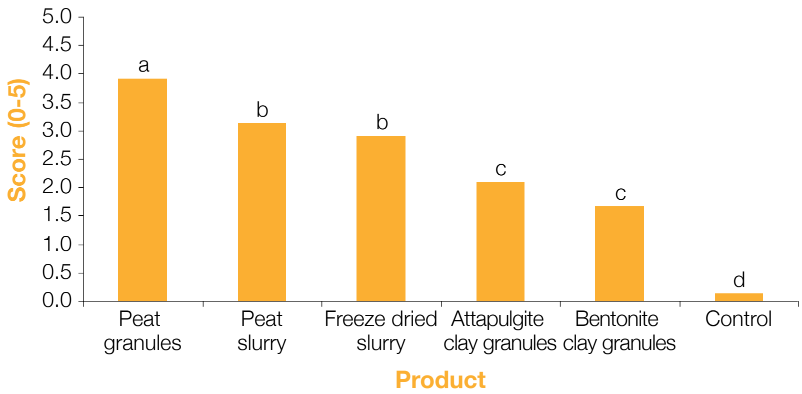


Figure 16: Effect of inoculant treatment on nodulation of chickpea roots, Narromine 2008.

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. This has been found in southern NSW, in work carried out by Denton *et al*. (2009). 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 2009 NSW Department of Primary Industries (NSW DPI)/Northern Grower Alliance (NGA) trial, nodulation from the slurry applied to seed method was significantly

More information

http://www.nga.org.au/results-and-publications/download/67/grdc-update-papers-general-/rhizobia-innoculation-methods-in-chickpeas/grdc-adviser-update-paper-goondiwindi-march-2011.pdf

http://www.grdc.com.au/GRDC-BPG-InoculatingLegumes

http://www.publish.csiro.au/paper/EA02218.htm

http://espace.library.uq.edu.au/view/UQ:259646

http://digital.library.adelaide.edu.au/dspace/handle/2440/40723

http://www.grdc.com.au/Media-Centre/Ground-Cover/Ground-Cover-Issue-105-July-August-2013/Trials-explore-chickpea-wheat-rotations

affected by fungicide (thiram + thiabendazole), where the fungicide and slurry were applied within 1 h 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. [14]

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 (e.g. chickpeas cropped straight into sorghum stubble), 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.

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. [15]

3.1.13 Inoculant and fungicide compatibility

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 (Table 5). [16]

Table 5: Effects of 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 plus 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 plus metalaxyl	106	173	279	54	1.8
Inoculum then metalaxyl	114	207	321	59	3.3
Metalaxyl 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. (NB inoculum plus fungicide is tank-mixed and applied as a single treatment.)

3.1.14 Compatibility with 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 (Table 6).

14 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-in-chickpeas/grdc-adviser-update-paper-goondiwindi-march-2011.pdf

15 R Brill, S Price (2012) Don't skimp on chickpea inoculation. *Australian Grain* Nov–Dec 2012, http://www.ausgrain.com.au/Back Issues/224ndgrn12/dont skimp on chickpea inoculation.pdf

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

Table 6: Rhizobial compatibility with different trace element products after 24 hours of tank mixing

TE formulation	Inoculant strain (by crop)				
	Field pea	Faba bean	Chickpea	Lupin	Soybean
Manganese 1	x	x	x	✓	✓
Manganese 2	✓	✓	✓	✓	✓
Zinc 1	x	x	x	x	✓
Zinc 2	x	x	x	x	✓
Zinc 3	x	x	x	x	✓
Zinc 4	x	✓	x	✓	✓
Zinc 5	x	✓	x	✓	✓

Source: Becker Underwood Pty Ltd.

Note the differences between inoculant types for a given trace element product, as well as differences between trace element products with a given inoculants. [17]

3.1.15 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 7).

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

Re-inoculate if not planted within 12 h of treatment. [18]

Table 7: Effects of 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 plus 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 plus Apron	106	173	279	54	1.8
Inoculum then Apron	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.

3.1.16 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.

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

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

- Use Group N chickpea inoculum
- Prepare slurry and apply in the shade, avoiding exposure to high temperatures (>30°C), direct sunlight, and hot winds.
- 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 h.
- Check air-seeders for excessively high temperatures in the air stream. Temperatures >50°C will kill the rhizobia. [19]

3.1.17 Rating nodulation and nitrogen fixation (effectiveness)

The amount of N fixed is strongly correlated with nodule rating as detailed in the following photo-standards (Figure 17).

When using this rating system, plants should be gently dug from the soil and the root system rinsed in water before scoring the level of nodulation.

Obvious signs of nodulation should be visible by 6 weeks after sowing (even in high soil-nitrate situations).

Rate the level of nodulation using the photo-standards provided. This is based on nodule number and their position on the root system.

Observe the pattern of nodules on the root system. Nodules on the main taproot clustered near the seed are a clear indication that nodulation occurred from the inoculation process. These are referred to as 'crown nodules'.

If there are no crown nodules, but nodules on the lateral roots, then it is likely that they have formed from native soil bacteria. These are usually ineffective in fixing N in chickpeas.

Nodules on both the crown and lateral branches indicate that inoculation was successful, and that bacteria have spread in the soil. The chickpea rhizobia are very aggressive and can spread short distances in the soil.

Inspect nodules for nitrogen fixation activity. The best method is to slice a few nodules open with a razor blade or sharp knife and look at their colour.

Young nodules are usually white and still need to develop. White nodules can also indicate the wrong bacteria in the nodule and these will not fix N.

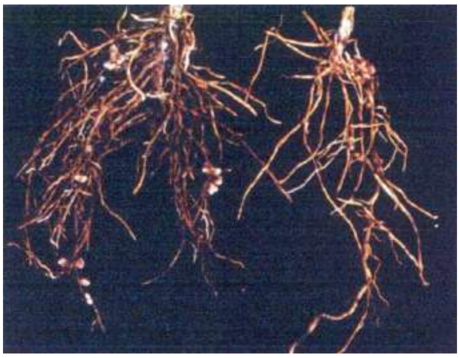
Effective nodules are a rusty red or pink colour inside and these usually are actively fixing N. Effective red nodules can sometimes turn green when a plant is under stress, such as from water or disease, or is suffering from nutrient deficiencies. These do not fix N, but they can change back to red-coloured and start to fix again if the stress is relieved without too much damage being done. Finally, black nodules are usually dead or dying. These are often seen as the crop matures, or after a crop has suffered severe water logging. [20]

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

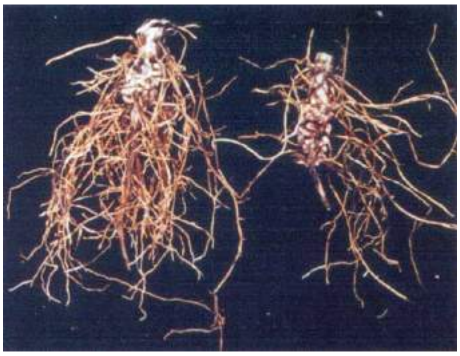
[20] Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

3.1.18 Key for assessing nodulation in winter pulse crops

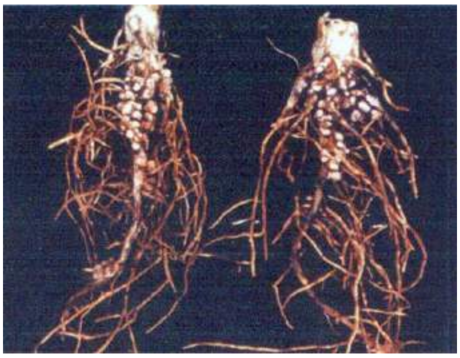
Figure 17 parts *a–f* show nodulation scores of 0–5, based on nodulation number and distribution where 0–1 is inadequate nodulation; 2–3 is adequate nodulation; and 4–5, good nodulation.



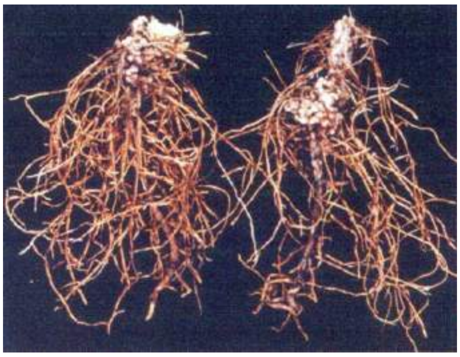
(a) Score 0: taproot, absent; lateral, absent/few.



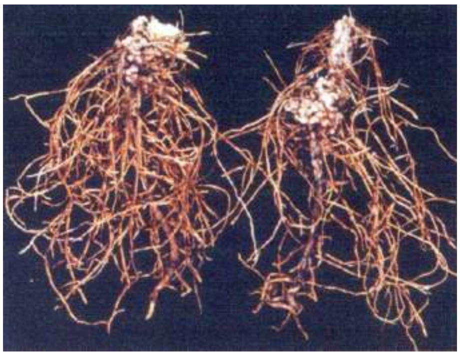
(b). Score 1: taproot, few/medium; lateral, absent.



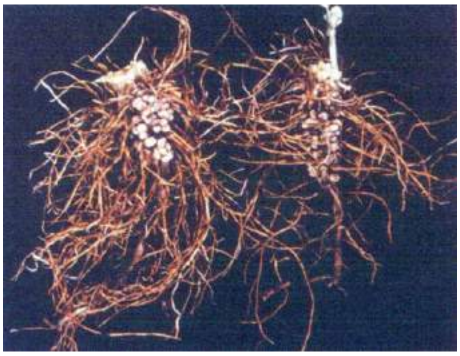
(c) Score 2: taproot, medium; lateral, absent/low.



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



(e) Score 4: taproot, high; lateral, medium.



(f) Score 5: taproot, high; lateral, high.

Figure 17: Nodulation scores based on number and distribution of nodules.

- Where plant-available soil N is low, the crop relies heavily on good nodulation for its N supply. A score of 4–5 is desirable.
- Where plant-available N is high, nodulation may be partly inhibited and the crop will depend mainly on the soil to supply N.
- A high score indicates that the crop will yield well and conserve soil N for use by a following crop.
- A low score suggests that the crop will yield poorly and deplete soil N. [21]

[21] TopCrop—Growers guide to assessing nodulation in pulse crops

More information

GRDC (2013), Clean seed and care the recipe for chickpea success

More information

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2012/04/Chickpea-seed-treatment-improves-crop-establishment-and-increases-yields-2011-trials-using-seed-from-2010-crops

More information

http://www.regional.org.au/au/asa/2012/crop-production/8197_haighb.htm

Pulse Australia (2016), Chickpea production: northern region

QDAF(2015) Planting chickpeas, Planting time

3.2 Seed treatments

No chickpea variety is resistant to seed infection by *Ascochyta* or *Botrytis*. All varieties will benefit from a seed dressing to protect against *Botrytis* and other seedling rots. Seed dressings may have a deleterious effect upon rhizobia, particularly under acid soil conditions, so minimize the contact time between these. Check the inoculum label. Apply seed dressing first then separately mix the inoculum and apply it to the seed immediately before sowing; or consider using granular or liquid injection inoculums. [22]

The use of P-Pickel T® or Thiraflo® is recommended for treating all planting seed throughout the northern region. These products are considered superior in minimising the risk associated with the spread of *Ascochyta* infection on seed. Seed retained on-farm should be from the cleanest paddocks, preferably where Ascochyta blight was not detectable in the previous season. [23]

NSW DPI trials clearly show that the fungus *Botrytis cinerea*, which causes chickpea pre- and post-emergence seedling disease, is readily controlled with registered seed treatments, provided they are applied correctly.

However, pathologists do not recommend using *Botrytis*-infected grain as planting material even if treated properly. The seed will have lower vigour and this will increase the risk of other seedling diseases, render weed management more difficult and may increase the risk of virus infection. Also, sowing rates will need to be increased to account for the reduced vigour, which may make using grower-retained seed uneconomical. [24]

3.3 Time of sowing

The key to planting chickpeas in the northern grains region 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. [25]

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.

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

22 W Hawthorne, J Davidson, L McMurray, K Hobson, K Lindbeck, J Brand (2012) Chickpea disease management strategy—southern region. Southern Pulse Bulletin 8, http://pulseaus.com.au/storage/app/media/crops/2012_SPB-Chickpea-disease-management.pdf

23 DAFF (2012) Planting chickpeas. Department of Agriculture, Fisheries and Forestry, 6 August 2012, http://www.daf.qld.gov.au/plants/field-crops-and-pastures/broadacre-field-crops/chickpeas/planting

24 K Moore, R Daniel, S Harden, A Mitchell, R Herron, P Nash, G Chiplin (2012) Chickpea seed treatment improves crop establishment and increases yields—2011 trials using seed from 2010 crops. GRDC Update Papers 10 April 2012, http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2012/04/Chickpea-seed-treatment-improves-crop-establishment-and-increases-yields-2011-trials-using-seed-from-2010-crops

25 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

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 (Figure 18)
- 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
- 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

Figure 18: Botrytis grey mould infected chickpeas. (Photo: Gordon Cumming, Pulse Breeding Australia)

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 (Table 8).

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 ≥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 <12°C at the placement depth. 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, 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.

Table 8: Preferred planting times for different regions

	April				May				June				July	
Week:	1	2	3	4	1	2	3	4	1	2	3	4	1	2
Central Queensland														
Maranoa–Balonne														
Western Downs														
Darling Downs														
Moree–Narrabri														
Walgett–Coonamble														
Liverpool Plains														
Central NSW (grey soil)														
Central NSW (red soil)														

Yellow boxes: marginal sowing time, increased costs and/or lower yields likely; green boxes, preferred sowing window

As the large-seeded Kabuli varieties mature later than the Desi varieties, they may need to be sown earlier than Desi in some districts. [26]

3.3.1 NSW DPI time-of-sowing trials

The two major constraints to chickpea production in the northern cropping region are disease and frost damage (Whish *et al* 2007). In each case, sowing date can influence yield by avoiding cold temperatures during flowering, and by reducing the impact of disease.

The optimal time to sow chickpea will depend on the interaction between the environment and the available varietal germplasm. Current chickpea genotypes have excellent frost tolerance when in the vegetative state, but conversely display one of the highest temperature thresholds for seed-set among cool-season (winter) pulse crops.

Mean daily temperature of <15°C has been shown to cause flower abortion (Clarke and Siddique 1998). Flowering initiation in chickpeas has been described as a photo-thermal response, but in most environments temperature is the main determinant. The optimum sowing date results in flowering when the risk of cold temperatures is low; it

More information

J Whish, B Cocks (2011). Sowing date and other factors that impact on pod-set and yield in chickpea

More information

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/02/Developing-a-plan-for-chickpeas-2013

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2012/04/Chickpea-time-of-sowing-trial-Trangie-2011

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2010/05/Chickpeas-in-2010-Low-temperature-effects-in-2009

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

is especially important to avoid frost during flowering, which can kill chickpea plants (Whish *et al*. 2007).

Choosing an optimum sowing time can also mean a compromise between maximising yield potential and minimising disease levels (Figure 19). Earlier sowing can expose the crop to more rain events, which can increase the risk of Ascochyta blight. It will also increase crop biomass, increasing the risk of Botrytis grey mould, lodging, and soil-moisture deficit during grain-fill. Later sowing can result in shorter plants (harvesting difficulties) and increased Heliothis pressure, but may reduce vegetative water use and reduce the exposure to Ascochyta and Phytophthora infection events and lessen the risk of Botrytis grey mould (Matthews and McCaffery 2011).

Figure 19: Choosing an optimum sowing time can be a compromise between maximising yield potential and minimising disease levels. (Photo: NSW DPI)

Chickpea time-of-sowing trials were conducted in 2010, 2011 and 2012 by NSW DPI at Trangie Agricultural Research Centre, to evaluate the impact of sowing date on phenology and yield of current and potential release cultivars. The 2010 trial succumbed to in-crop waterlogging and wet weather at harvest and it was not harvested.

The time of sowing of chickpea has been debated as an issue at GRDC Northern Update sessions. The development and release of new chickpea varieties with high-yielding attributes (largely due to greater regional adaptation and improved disease tolerance) has led to the belief that early sowing (early May) would be the key to optimising water use, through the development of increased biomass and hence earlier flowering.

This trial has shown that earlier flowering does not necessarily translate into higher yield, due to the effect of lower temperatures during early flowering and a greater potential risk of disease. Conversely there is also a yield penalty from later sowing (late June), but these chickpea plants are able to compensate to some effect compared with very early sowing. Further research will be conducted over several seasons to develop sound recommendations for the region. [27]

3.3.2 Time-of-sowing trial results 2012

Chickpea time-of-sowing trials in both 2011 and 2012 were conducted with a full soil-moisture profile at planting. The 2011 season was characterised by wet conditions post planting in May, resulting in an increased incidence of Phytophthora root rot, followed

More information

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2012/04/Chickpea-time-of-sowing-trial-Trangie-2011

http://www.dpi.nsw.gov.au/agriculture/broadacre/guides/winter-crop-variety-sowing-guide

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/02/Virus-in-chickpea-in-northern-NSW-2012

[27] L Jenkins, R Brill (2012) Chickpea time of sowing trial, Trangie 2011. GRDC Update Papers 10 April 2012, http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2012/04/Chickpea-time-of-sowing-trial-Trangie-2011

by a dry winter and spring. Chickpea foliar diseases did not affect yield in either trial due to fungicide applications and the dry conditions in July–September.

The 2011 trial showed a significant yield penalty from early sowing (5 May). This was due to both the increased incidence of Phytophthora root rot and the effect of low temperatures on pod development. Optimum yields were achieved from mid-season sowing (18 May and 9 June). The late sowing (27 June) in 2011 had lower yields than the two mid-season sowings, but still yielded higher than the early sowing.

The 2012 trial confirmed the results from 2011 (i.e. mid May to early June remains the optimum period to plant most current chickpea varieties with Jimbour-type maturity, e.g. PBA HatTrickⓓ and PBA Boundaryⓓ, in the central-western region (Table 9)). Early planting (early May) can increase the risk of exposure to disease infection events, as occurred in 2011 (resulting in lower yields), although this did not occur in 2012. Planting chickpeas in mid-late June resulted in significantly lower yields in both the 2011 and 2012 trials.

In 2011, PBA HatTrickⓓ was the overall highest yielding variety (mean yield 1.37 t/ha), although not significantly higher than PBA Boundaryⓓ. In 2012, PBA Boundaryⓓ was the highest yielding variety (mean yield 1.62 t/ha), and higher yielding than PBA HatTrick ⓓ (1.5 t/ha). This reinforces the view that while Jimbourⓓ-type maturities are ideal for the central-west of NSW, each year will be slightly different in terms of variety response to the season. Knowledge of soil type and paddock disease risk will assist in choice of variety, with PBA Boundaryⓓ not recommended in paddocks known to have a history of Phytophthora root rot.

In 2012, the seeding rate component of the trial showed that targeting a lower plant population (15 plants/m^2) reduced yield potential, regardless of sowing time. Targeting a higher plant population (45 plants/m^2) had higher yields than the 15 and 30 plants/m□ treatments at all sowing times. [28]

Table 9: Grain yield (t/ha) of seven chickpea varieties sown at four sowing times at Trangie Agricultural Research Centre, 2012

Variety	9 May	21 May	1 June	20 June	Mean of variety
CICA-0912	1.44	1.56	1.50	1.24	1.43
Flipperⓓ	1.65	1.55	1.45	1.13	1.44
Genesis™ 090	1.69	1.73	1.65	1.25	1.58
Genesis™ Kalkee	1.42	1.64	1.25	0.81	1.28
PBA Boundaryⓓ	1.60	1.71	1.76	1.42	1.62
PBA HatTrickⓓ	1.51	1.68	1.46	1.35	1.50
Sonaliⓓ	1.55	1.52	1.41	1.09	1.39
Mean of sowing time	1.55	1.63	1.50	1.18	1.46
l.s.d. (*P* = 0.001)	Variety = 0.108 t/ha, sowing time = 0.08 t/ha				

More information

http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0004/468328/Northern-grains-region-trial-results-autumn-2013.pdf

3.3.3 Frost damage

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.

[28] L Serafin, S Simpfendorfer, M Gardner, G McMullen (Eds) (2013) Northern grains region trial results, autumn 2013, DPI NSW, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0004/468328/Northern-grains-region-trial-results-autumn-2013.pdf

Damage to flowers and pods:

Freezing temperatures destroy flowers and young developing seed (Figure 20). 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.

Figure 20: Frosted chickpea crops at flowering.

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

Simulated frost risk for Amethyst planted across a range of sowing dates is presented in Table 10.

Values in the table are the percentage chance of receiving a –1°C frost or colder (at screen height) during late flowering–early pod-fill when combined with dry soil conditions that would limit further flowering.

The results show that:

- Frost risk is much higher in southern districts than in central Queensland
- Frost risk declines with later sowing dates
- Frost risk could be minimised to ≤10% by sowing:
- no earlier than mid-April in central Queensland
- no earlier than late May on the eastern Downs
- no earlier than mid-May for western Downs and Maranoa. [29]

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

Table 10: Percentage chance of receiving a frost during late flowering–early pod-fill combined with dry soil conditions

Location	Sowing date						
	15 April	1 May	15 May	1 June	15 June	1 July	15 July
Emerald	10	3	3	0	0	0	0
Dalby	71	35	29	10	3	3	0
Roma	87	35	13	6	3	0	0
Goondiwindi	35	13	10	3	0	0	0

Source: M Robertson, CSIRO.

3.4 Seeding rates

Yields are relatively stable within the range 20–30 plants/m^2; however, populations of 25 plants/m^2 will optimise yields in the northern region. Research has shown that slightly higher populations are required in relatively colder production areas in northern NSW.

Higher populations are justified for late plantings, while lower populations of about 20 plants/m^2 are often recommended for crops grown in wide row spacings (1 m). High populations planted in wide rows often result in thin main stems and a higher risk of lodging.

Seeds are not all equal, and some grow better than others. Before deciding on a seeding rate, take a representative sample and have it germination-tested.

Seeding rates can have a very significant effect on crop yields; however, there are considerable differences in seed size between individual pulse varieties.

When determining a seeding rate, consider 'plant population size' and not just kg of seed per ha. In other words, the kg rate should be adjusted to achieve a target population of plants based on seed size and germination percentage.

Calculating seeding rates

Seeding rate for the target plant density can be calculated using germination percentage, 100-seed weight and establishment percentage.

$$\text{Seeding rate (kg/ha)} = \frac{\text{100 seed weight (g) x target plant population per m}^2\text{ x 1000}}{\text{germination\% x estimated establishment\%}}$$

Example

100-seed weight = 20 g

Target plant density = 25 plants/m^2 (i.e. 250,000 plants/ha)

Germination% = 95%

Estimated establishment% = 80% (Note: an establishment percentage of 80–90 % is a reasonable estimate, unless sowing into adverse conditions.)

Seeding rate (kg/ha) = (20 x 25 x 1000)/(95 x 80) = 65 kg/ha

To determine seed weight, weigh 100 seeds (g).

If you have seeds per kg from a laboratory test, this can be easily converted to 100-seed weight, as follows:

100-seed weight = (1000/seeds per kg) x 100

Note: Optimum plant populations vary with the growing location and the pulse crop and variety being sown (Table 11). [30]

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

More information

[Pulse Australia (2016), Chickpea production: northern region](#)

L Jenkins, A Verrell (2015), Chickpea and mungbean research to maximise yield in northern NSW

K McKenzie, H Cox, R Rachaputi, B Raymond, N Seymour (2014), Impact of row spacing and populations on chickpeas

R Raymond, K McKenzie, R Rachaputi (2015), Impact of row spacing on chickpea fababean and mungbean

M Thomson (2014), New cultivars and narrow rows boost Darling Downs chickpea yields

K McKenzie, N Seymour, H Cox, B Raymond, RCN Rachaputi (2015), Chickpea and faba bean agronomy ideal row spacing and populations

A Verrell, L Jenkins (2014), Effect of row spacing on yield in chickpea under high yield potential

A Verrell, R Brill, L Jenkins (2014), The effect of plant density on yield in chickpea across central and northern NSW p39

Table 11: Seeding rate (kg/ha) required for targeted plants per 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™425	Small Kabuli	29	76	95	114
Genesis™Kalkee	Larger Kabuli	46	121	151	182
Flipper⦸	Medium Desi	18	47	59	71
Genesis™509	Small Desi	16	42	53	63
Genesis™510	Small Desi	16	42	53	63
Genesis™836	Medium Desi	18	47	59	71
Kyabra⦸	Large Desi	25	66	82	99
PBA Boundary⦸	Medium Desi	20	53	66	79
PBA HatTrick⦸	Medium Desi	21	55	69	83
PBA Slasher⦸	Medium Desi	20	53	66	79
Yorker⦸	Medium Desi	21	55	69	83

3.5 Targeted plant population

Planted on a row crop configuration (up and back on 50–100-cm rows), chickpeas can benefit from a reduced incidence of Ascochyta blight by improving airflow between the rows.

The wider row spacing can reduce spray costs by allowing for banded spraying. Harvesting height is improved if the chickpeas are sown on the inter-row, between the rows of the previous cereal crop. This enables the standing stubble to act as a trellis.

Newer chickpea varieties with genetic resistance to Ascochyta blight may result in a re-evaluation of row spacing for this crop in the future. [31]

When sowing within the optimum sowing window mid-May–mid-June:

- For yield potential ≥1.5 t/ha, sow at ≥25 plants/m².
- For yield potential ≤1.5 t/ha, sow at ≥20 plants/m².

When sowing very late, sow at high plant density to reduce losses due to viruses; do not sow < 20 plants/m² (Table 12). [32]

Recent NSW DPI research builds on research work from the early 1990s, which gave rise to the rule of thumb commonly used in northern NSW and Queensland (i.e., that row spacing ranging from 25 to 75 cm results in no yield difference).

Chickpeas are successfully grown using a wide range of row spacings, from 20 to 100 cm, with wider rows (50–100 cm) becoming quite common. There was a need to look at the effect of row spacing under situations of high yield potential, with current varieties and newer agronomic practices.

The research shows that new varieties such as PBA HatTrick⦸ have a lower rate of yield decline at wider row spacing than older varieties such as Amethyst. Researchers

[31] GRDC (2011) Northern region—A systems approach to row spacing. GRDC Factsheet January 2011, https://www.grdc.com.au/Resources/Factsheets/2011/02/Crop-Placement-and-Row-Spacing-Northern-Fact-Sheet

[32] A Verrell, R Brill, L Jenkins (2014) The effect of plant density on yield in chickpea across central and northern NSW. GRDC Update Papers 5 March 2014, http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/The-effect-of-plant-density-on-yield-in-chickpea-across-central-and-northern-NSW

More information

http://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Effect-of-row-spacing-on-yield-in-chickpea-under-high-yield-potential

http://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/The-effect-of-plant-density-on-yield-in-chickpea-across-central-and-northern-NSW

https://www.grdc.com.au/Resources/Factsheets/2011/02/Crop-Placement-and-Row-Spacing-Northern-Fact-Sheet

recommend the following rules of thumb when sowing within the optimum window of mid-May–mid-June under conditions of high yield potential:

- For yield potential ≥2.0 t/ha, sow on narrow rows (≤40 cm).
- For yield potential ≤2.0 t/ha, row spacing has less of an impact on yield.
- When sowing very late, sow on narrow rows at adequate plant density.
- When sowing very early, sow on wider rows to reduce early soil water extraction.

Table 12: Effect of sowing date and row spacing on the yield of different varieties over consecutive seasons.

Year	Sow date	Variety	Row spacing	Yield	Standard error
2008	30 May	Flipper	40 cm	2.08	± 0.162
		Flipper	80 cm	1.79	
		Jimbour	40 cm	2.31	
		Jimbour	80 cm	1.83	
2009	29 May	Flipper	40 cm	2.70	± 0.190
		Flipper	80 cm	2.25	
		Jimbour	40 cm	2.83	
		Jimbour	80 cm	2.23	
2010	1 June	Amethyst	40 cm	2.58	± 0.093
		Amethyst	80 cm	2.14	
		PBA HatTrick	40 cm	2.98	
		PBA HatTrick	80 cm	2.74	
2013	22 June	PBA HatTrick	40 cm	2.20	± 0.033
	17 July	PBA HatTrick	40 cm	1.45	± 0.028
		PBA HatTrick	80 cm	1.08	

All sowings at a fixed plant density of 30 plants/m²

The significant yield advantage of narrow rows over wide rows for the very late sown (17 July) crop supports the findings of Whish and Cocks (2004) and Whish (2007), in which narrow-planted late crops produced higher yields. [33]

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 4 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

[33] A Verrell, L Jenkins (2014) Effect of row spacing on yield in chickpea under high yield potential. GRDC Update Papers 5 March 2014, http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Effect-of-row-spacing-on-yield-in-chickpea-under-high-yield-potential

More information

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/02/Row-placement-strategies-in-a-break-crop-wheat-sequence

- better protection to break-crop seedlings from standing wheat stubble [34]

3.7 Sowing depth

Chickpeas should be sown 5–7 cm deep into good soil moisture.

The seedlings are robust, provided high-quality seed is used. Sowing at 5–7 cm has several agronomic advantages, as it:

- reduces the risk of damage from pre-emergent residual herbicides (e.g. simazine, Balance®);
- promotes the early formation of lateral roots in the topsoil;
- enhances inoculum survival in moist soil; and
- eliminates a significant proportion of Ascochyta-infected seeds (due to mortality of diseased seed).

Press-wheels can improve establishment, although heavy pressures should be avoided.

V-Shaped press-wheels will leave a furrow down the planting line, which can lead to concentration of residual herbicides in the furrow after rainfall, and subsequent crop damage.

3.7.1 Deep planting

Deep planting is not only an extremely valuable tool under drought conditions, but can also offer major advantages in most years including:

- planting at the optimum time
- freeing up valuable time for planting wheat when suitable planting rains do fall
- avoidance of residual herbicide damage
- better development of lateral roots
- improved nodulation

Many growers have been deep-planting chickpeas for some time. Excellent plant emergence has been achieved from up to 15 cm deep, and planting depth can be varied from 5 to 20 cm according to seasonal conditions.

There are a few key points to remember:

- Plan ahead when deep planting. It pays to plant early in the planting window to allow for any delay in emergence (typically 10–14 days), ensuring that the plant is able to grow tall enough to facilitate harvest.
- Ensure you have high quality planting seed. Check your germination percentage, vigour and seed counts (seeds/kg) and adjust seeding rates accordingly.
- When deep planting, plan levelling of the paddock after planting to reduce the risk of herbicide damage when using a pre-emergent herbicide such as simazine and/or Balance®.
- Decide on a planting depth to ensure that all seeds are planted into moisture. Experience shows that it is better to err on the deep side rather than planting too shallow into marginal moisture.
- To maximise yield potential, paddocks should be selected carefully to avoid any subsoil constraints, such as salinity, to ensure that the crop can gain maximum access to all the stored soil moisture and nutrients. [35]

[34] A Verrell (2013) Row placement strategies in a break crop wheat sequence. GRDC Update Papers 26 Feb 2013, http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/02/Row-placement-strategies-in-a-break-crop-wheat-sequence

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

3.8 Sowing equipment

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.

3.8.1 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.

3.8.2 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. Figure 21 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.

Airseeders with large, single fluted rollers cannot meter faba and broad beans >18 mm without modifications to the metering roller. Consult the dealer about possible modifications.

Significant levels of seed damage can be caused in airseeders by excessive air pressure, so be careful to use only enough air to ensure reliable operation.

Figure 21: 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, http://www.grdc.com.au/uploads/documents/3 Seeding.pdf)

3.8.3 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 (Figure 22) (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.

A comparison of the key functions that are critical for seed drills and no-till is shown in Table 13.

In interpreting the functions listed in Table 13 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 (Figure 23). These tines do perform differently in some functions in Table 13.
- 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 (Figure 24).
- 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.

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 faba and broad beans or Kabuli chickpeas (Figure 25).

Table 13 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 (Figure 26).

Figure 22: One of several seeding mechanisms for uniform sowing depth using the press wheel for depth control.

Figure 23: 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.

Figure 24: The DBS system parallelogram for uniform seeding depth and deep placement of seed or fertiliser.

Figure 25: A 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.

Figure 26: A Case IH SDX-40 single-disc drill.

Table 13: 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
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 fertilizer banding is effective with high analysis fertilizers	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

	Narrow point	Wide point	Sweep	Double disc	Single disc	Slanted disc	Combined winged tine & disc[B]
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

Source: C.J Baker (2010), SANTFA 12th Annual Conference pp. 7–13.
[A]Functions that may be deleted in some circumstances, but all other functions are universal.
[B]Combination is otherwise known as the Cross Slot™ or Bio Blade.

In Table 13, 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 13 as a guide only to help select your own openers to suit your conditions and circumstances. [36]

[36] Pulse Australia (20130 Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

SECTION 4

Plant growth and physiology

4.1 Key to growth stages

The chickpea growth stages key is based on counting the number of nodes on the main stem.

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 node counted must have its leaves unfolded.

The R stages begin when the plant begins to flower at any node.

Germination is hypogeal, with germination occurring when the cotyledons are below the soil surface. This enables the seedling to emerge from sowings as deep as 15 cm. In arid regions, chickpea is sown deep because surface moisture is often inadequate to assist crop establishment.

The node at 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 1–8 primary branches on the main stem, depending on growing conditions).

A node is counted as developed when 6–15 leaflets have reached the stage where they are unfolded and flattened out (Figure 1).

Figure 1: Growth habit of a chickpea plant.

Chickpeas are considered very indeterminate in their growth habit (i.e. their terminal bud is always vegetative and keeps growing). Vegetative growth continues even as the plant switches to reproductive mode and flowering begins (Table 1).

Flower terminals normally develop from the axillary bud at the base of each node. Chickpea flowers are purple in the Desi type and white to cream in the Kabuli type. Flowers are borne on a jointed peduncle that arises from nodes. Flowers are primarily self-pollinated, with most reports measuring 100% self-pollination.

Table 1: 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
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

More information

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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. [1]

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

More information

[J Whish (2016), Accessing and using day degrees in field crops as a tool to assist crop management]

[QDAF (2015), Planting chickpeas, Effects of temperature and frost damage]

4.2 Crop growth and development

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 semi-arid 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.

Crop duration is highly correlated with temperature, such that crops will take different times from sowing to maturity under different temperature regimes. The concept of thermal time is the mechanism used to represent a crop's evolved requirement to accumulate a minimum time for development through each essential growth stage (e.g. vegetative or reproductive growth). Consequently, crops growing under low air temperatures generally require more time to develop than crops growing under warmer temperatures. Thermal time is also referred to as heat units, day-degrees or growing degree-days (sometimes represented as °Cd).

The base temperature for calculating thermal time for chickpea is 0°C.

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 2 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 2). Chickpea is a photoperiod-sensitive, long-day plant, where flowering is delayed as day length becomes shorter than a base photoperiod (17 h). [2]

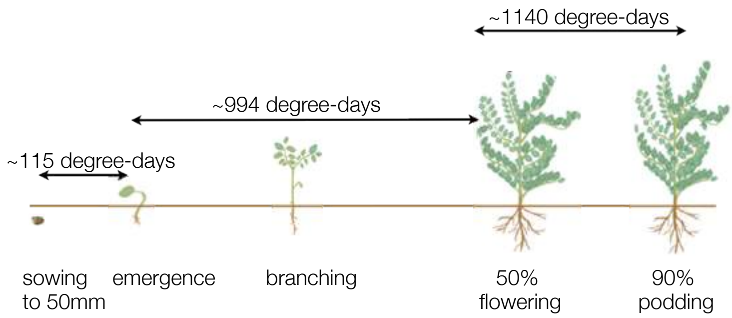


Figure 2: Key developmental stages of chickpea and their thermal time targets. (Source: J. Whish, CSIRO)

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

4.2.1 Emergence

Pulses are classed as 'epigeal' if the cotyledons appear above the ground or 'hypogeal' if they remain below the ground. Chickpeas are hypogeal (Figure 3). Seedlings with hypogeal emergence are less likely to be killed by frost, wind erosion or insect attack because new stems can develop from buds at nodes, at or below ground level. By contrast, if an epigeal pulse is broken below the cotyledons, the plant will die, as there are no buds from which to shoot.

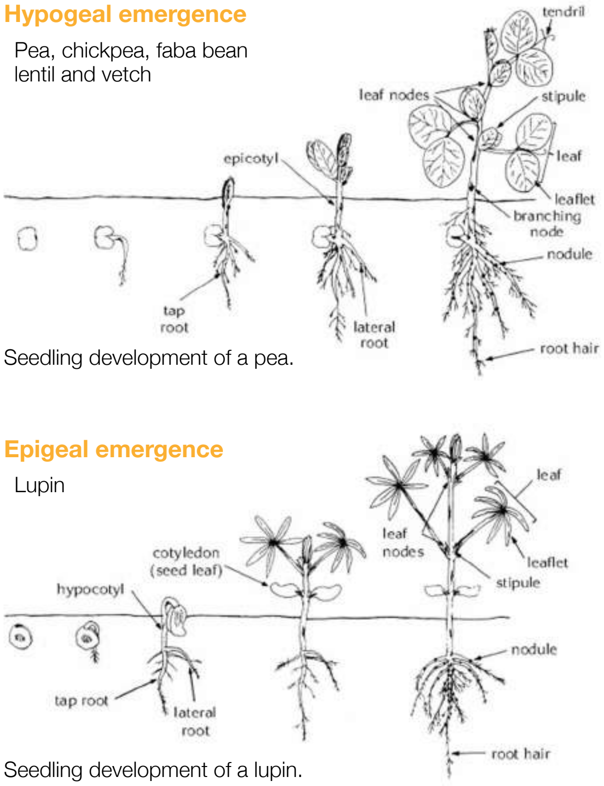


Figure 3: Hypogeal and epigeal emergence patterns in pulses.

Under optimum moisture and temperature conditions, chickpea seeds imbibe water quickly and germinate within a few days, providing temperatures are >0°C. Unlike lupins, chickpea seedlings have hypogeal emergence, that is, their cotyledons (embryonic leaves) remain underground inside the seed coat while providing energy to the rapidly growing roots and shoots.

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 (Figure 4). [3]

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

Figure 4: Chickpea growth and development from germination to 2 months. Plants may vary according to variety and environment. (Photo: H. Clarke, UWA)

4.2.2 Leaves

Leaves in chickpea are alternate along the branch (Figure 5). Each leaf is composed of 10–16 serrated leaflets, which can fold slightly in dry conditions to minimise transpiration. Despite having more leaves and branches than other legume crops such as faba bean, canopy development in chickpea is slow, especially during the cool winter months.

Figure 5: 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, is densely covered with fine hairs known as trichomes (Figure 6). Many are glandular and secrete a highly acidic substance containing mainly malic acid but also some oxalic and citric acid. Secretions of acid increase with temperature throughout the day; they are diluted when the hairs are washed by dew overnight, and by wind shaking the acid droplets from the hairs.

Figure 6: A green pod covered in glandular hairs excreting acid. (Photo: H. Clarke, UWA)

The acid seems to play a role in protecting the plant against pests such as redlegged earth mite, lucerne flea, aphids and pod borers. The acid is also secreted through the root system and it can solubilise soil-bound phosphate and other nutrients. The acid also corrodes leather boots. [4]

4.2.3 Roots

Chickpea root systems are usually deep and strong, and contribute to the ability to withstand dry conditions (Figure 7). 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; however, subsoil constraints such as soil chloride >800 mg/kg soil in the top 60 cm will restrict root growth and water availability.

As well as their role in water and nutrient uptake, chickpea roots develop symbiotic nodules with the *Rhizobium* bacteria, capable of fixing atmospheric nitrogen (N2). The plant provides carbohydrates for the bacteria in return for nitrogen fixed inside the nodules.

These nodules are visible within about 1 month after plant emergence, and eventually form slightly flattened, fan-like lobes (Figure 8). Almost all nodules are confined to the top 30 cm of soil and 90% are within 15 cm of the surface. When cut open, nodules actively fixing nitrogen have a pink centre (Figure 9). Nitrogen fixation is highly sensitive to waterlogging, and hence, chickpea needs well-aerated soils. [5]

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.

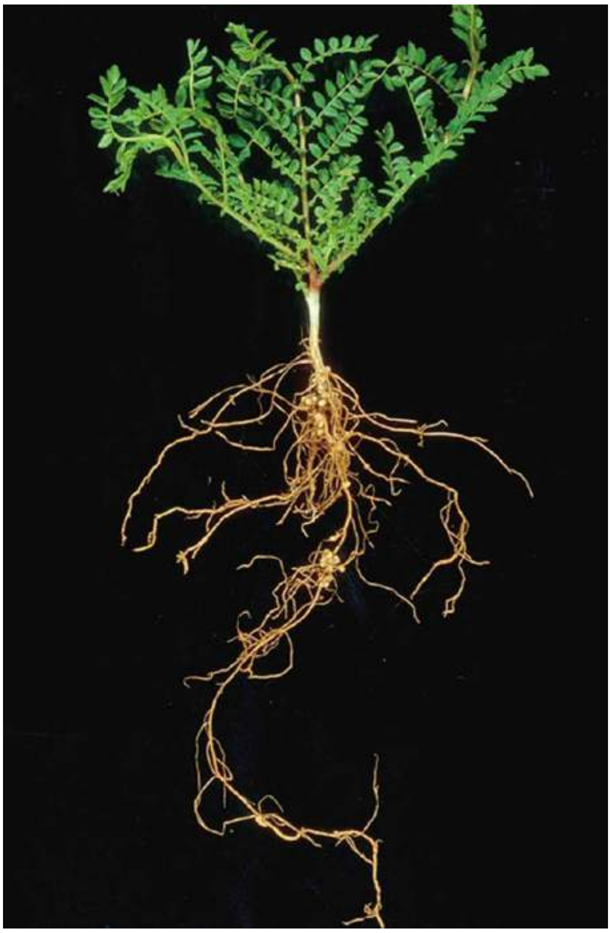


Figure 7: Chickpea usually has a deep tap root system. (Photo: P. Maloney, DAFWA)

Figure 8: Nodulated chickpea roots. (Photo: G. Cumming, Pulse Australia)

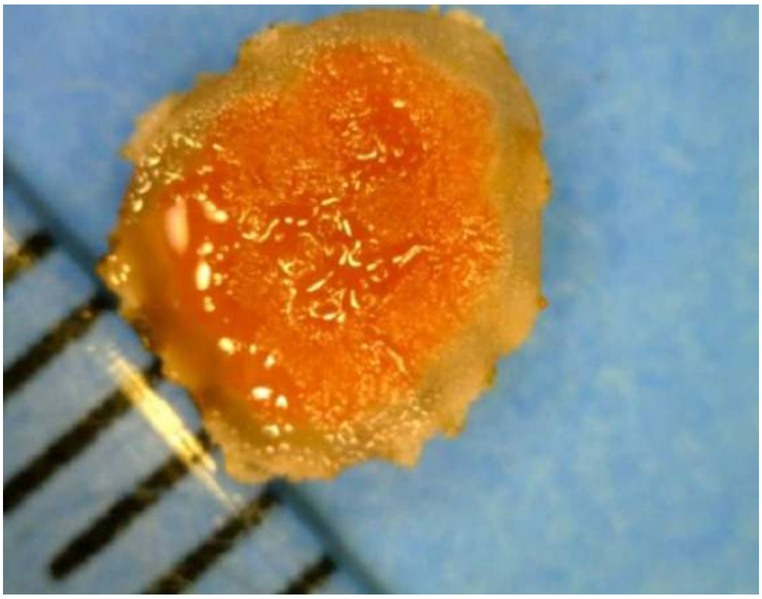


Figure 9: Active nodules have a pink centre. (Photo: G. Cumming, Pulse Australia)

4.2.4 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 10). The main stem and branches can attain a height of about 40–100 cm. Kabuli varieties are generally taller than Desi varieties.

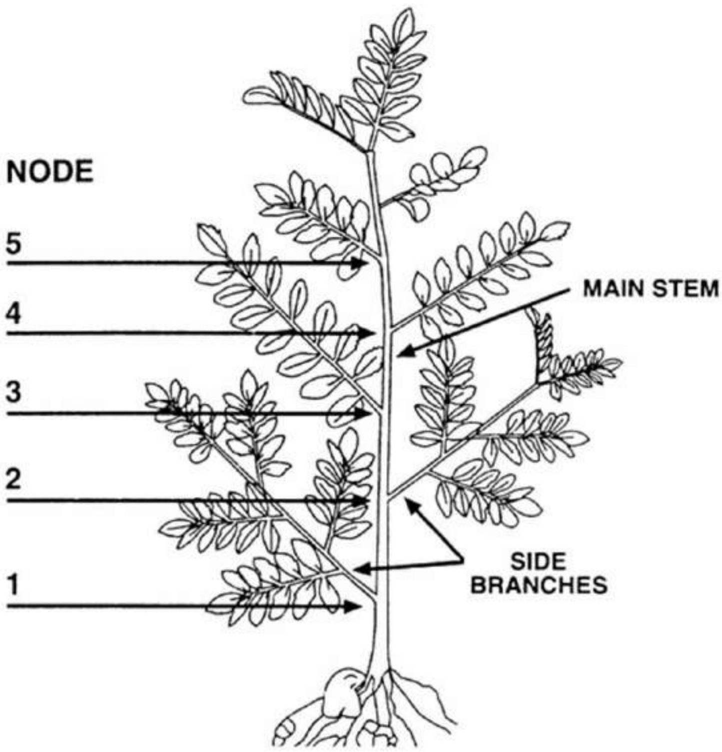


Figure 10: Chickpea at the 5–7-node stage of development.

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 chickpea, 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. [6]

4.2.5 Flowering, podding and seed development

Growth in chickpea 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 (Figure 11).

Figure 11: Different stages of flower development on the same chickpea branch. (Photo: K. Siddique, DAFWA)

The duration of vegetative growth before flowering depends on many factors. 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.

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

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 below). In general, warmer temperatures hasten development.

Petals are generally purple in the Desi type (Figure 12) and white to cream in the Kabuli type. Purple-flowered Desi types generally contain high amounts of the red pigment anthocyanin, and their leaves, stems and seed coats are generally dark.

Figure 12: Desi chickpea purple flowers. (Photo: G. Cumming, Pulse Australia)

By contrast, the white-flowered Kabuli types lack anthocyanin, have light green leaves and stems, and pale seeds (Figure 13). Increased pigmentation is evident following environmental stresses such as low temperature, salinity, waterlogging, drought, and virus infection, especially in Desi types.

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. Natural cross-pollination has been reported; however, most studies indicate 100% self-pollination.

Figure 13: Kabuli chickpea lack anthocyanin, hence their white flowers. (Photo: G. Cumming, Pulse Australia)

Chickpea plants generally produce many flowers. However, about 30% do not develop into pods, depending upon the variety, sowing date and other environmental conditions.

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 6 days. The seed then fills over the next 3–4 weeks (Figure 14). 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.

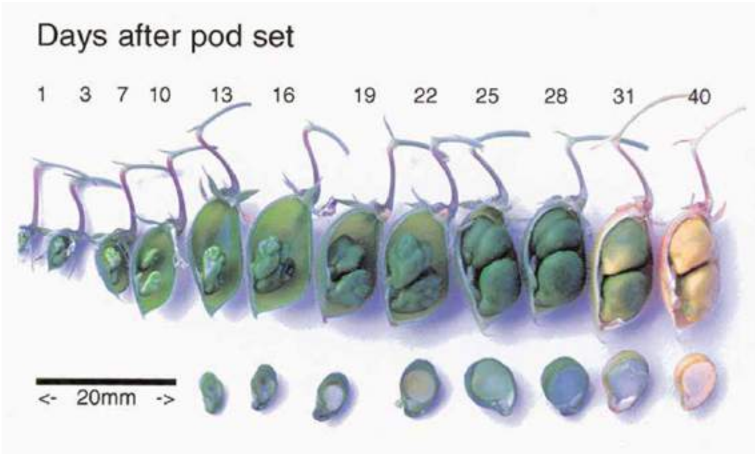


Figure 14: Development of pods and seeds from pod to maturity. (Photo: L. Leport)

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. [7]

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

4.2.6 Climatic requirements for flowering

The timing of flowering is an important trait affecting the adaptation of crops to low-rainfall, Mediterranean-type environments (such as the south-west of Western Australia), and seed yields of many crops in these areas have been increased by early sowing and the development of early-flowering varieties.

Temperature, daylength, and drought are the three major factors affecting flowering in chickpea. Temperature is generally more important than daylength. Flowering is invariably delayed under low temperatures but more branching occurs.

Progress towards flowering is rapid during long days, whereas under short days (>17 h daylight), flowering is delayed but never prevented.

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 (Singh 1996). The critical mean or average daily temperature for abortion of flowers in most current varieties is <15°C (Siddique 1998). Abortion occurs below this temperature because the pollen becomes sterile and reproductive structures do not develop. Flowers may develop below this temperature but they contain infertile pollen.

Once true flowers are produced, a period of cool weather can cause some flower or pod abortion. Figure 15 provides an example of when effective seed set commenced at Trangie, NSW, in 2008. If flowering starts before average daily temperatures reach 15°C, then flowers will continue to abort until temperatures increase beyond this critical level.

Selection of sowing date is a trade-off between sowing early with high yield potential in those years with a warmer spring and lower yield potentials with delayed sowing to ensure that flowering occurs with temperatures closer to 15°C in cooler springs.

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.

Note that 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).

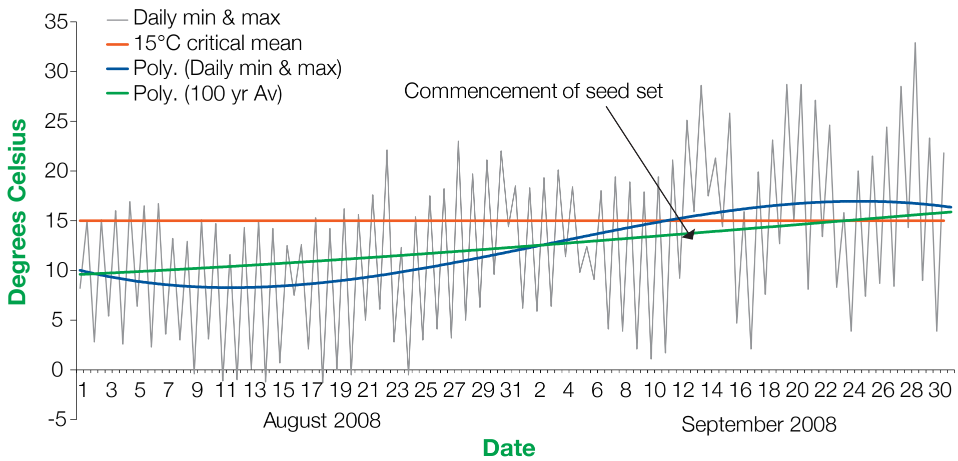


Figure 15: Commencement of effective seed set, showing minimum, maximum and average daily temperatures at Trangie NSW, 2008.

In addition to the effects of cold described above, 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 (Figure

16). However, chickpea has an excellent ability to recover from this superficial damage and is able to regenerate new branches in severe cases.

Figure 16: Frost can cause bends like a hockey stick in chickpea stems. (Photo: S. Loss, DAFWA)

Late frosts also cause flower, pod and seed abortion (Figure 17). 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.

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. Minimum temperatures <5°C during the reproductive stage will kill the crop, but new regrowth can occur from the base of the killed plants if moisture conditions are favourable.

Figure 17: 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)

More information

Paper in the journal *Crop & Pasture Science*: High temperature tolerance in chickpea and its implications for plant improvement

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.

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. [8]

4.2.7 Tolerance to low temperature

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 found. Some genotypes from India are less sensitive than those currently grown in Australia, and these are being utilised in chickpea-breeding programs at Department of Agriculture and Food Western Australia (DAFWA) and the University of Western Australia.

Controlled environment studies at University of Western Australia have identified two stages of sensitivity to low temperature in chickpea. The first occurs during pollen development in the flower bud, resulting in infertile pollen even in open flowers. The second stage of sensitivity 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 (Figure 18).

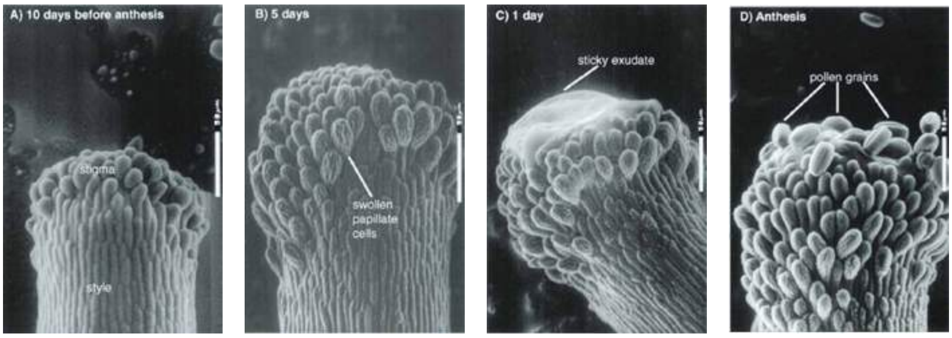


Figure 18: Development of the style and stigma of chickpea flowers taken with an electron microscope. (Photo: H. Clarke, UWA)

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 (Figures 19 and 20).

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

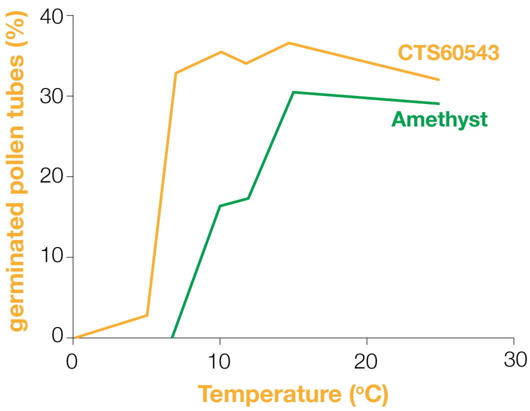


Figure 19: Proportion of pollen germination at various temperatures in cold-sensitive (Amethyst) and cold-tolerant (CTS60543) varieties.

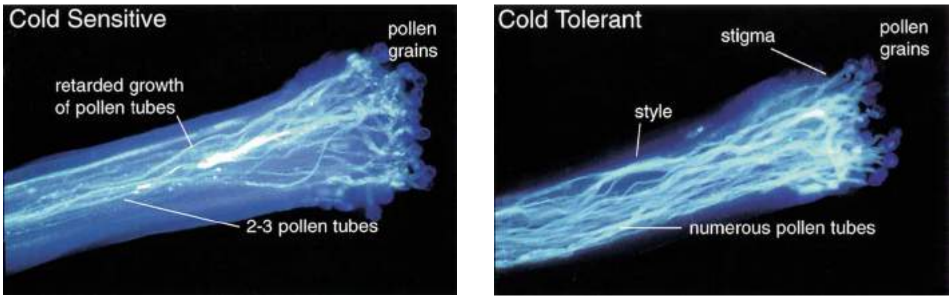


Figure 20: Pollen tube growth (stained with a fluorescent dye) in the stigma of cold-tolerant and cold-susceptible chickpea varieties. (Photo: H. Clarke, UWA)

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 about 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. [9]

4.2.8 Maturity

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 (Figure 21). This is typical of grain legumes and annual plants in general.

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.

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

Figure 21: Chickpea may be forced to mature early on soils with poor moisture holding capacity. (Photo: K. Siddique, DAFWA)

As leaves begin to senesce, there is a rapid re-translocation of dry matter from leaves and stems into the seeds.

Recent 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.

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 (Figure 22). [10]

Figure 22: Physiologically mature grains 'rattle pod'. (Photo: G. Cumming, Pulse Australia)

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

4.3 Flowering and maturity date in chickpea

Table 2 shows the average date for flowering to commence on 50% of plants and for maturity (pods brown) to occur on 95% of plants in a paddock for a range of sowing dates and locations in the northern region. Planting date has a major impact on actual crop yields, because the flowering date will determine whether fertile flowers are produced and pods will form. [11]

Table 2: Average flowering and maturity dates (cv. Amethyst) in relation to planting across a range of areas

Sowing date	Dalby	Goondiwindi	Roma	Emerald	Walgett	Narrabri	Dubbo
Flowering date (50% flower)							
1 May	18 July	18 July	17 July	30 June	6 Aug.	7 Aug.	7 Aug.
15 May	5 Aug.	5 Aug.	4 Aug.	18 July	12 Aug.	13 Aug.	24 Aug.
1 June	23 Aug.	22 Aug.	20 Aug.	5 Aug.	25 Aug.	27 Aug.	8 Sept.
15 June	3 Sept.	3 Sept.	31 Aug.	19 Aug.	8 Sept.	7 Sept.	18 Sept.
1 July	14 Sept.	14 Sept.	11 Sept.	31 Aug.	17 Sept.	18 Sept.	27 Sept.
15 July	22 Sept.	22 Sept.	20 Sept.	10 Sept.	26 Sept.	26 Sept.	4 Oct.
Maturity date (90% brown pods)							
1 May	9 Oct.	9 Oct.	5 Oct.	12 Sept.	14 Oct.	14 Oct.	29 Oct.
15 May	20 Oct.	19 Oct.	15 Oct.	25 Sept.	25 Oct.	24 Oct.	7 Nov.
1 June	31 Oct.	30 Oct.	26 Oct.	8 Oct.	4 Nov.	4 Nov.	16 Nov.
15 June	8 Nov.	6 Nov.	2 Nov.	17 Oct.	10 Nov.	10 Nov.	22 Nov.
1 July	15 Nov.	14 Nov.	10 Nov.	26 Oct.	17 Nov.	17 Nov.	29 Nov.
15 July	21 Nov.	20 Nov.	16 Nov.	3 Nov.	22 Nov.	22 Nov.	4 Dec.

(Source: J. Whish, CSIRO.)

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

4.3.1 Effect of planting time on flowering in different areas of Queensland

The full set of values for all regions can be found in Appendix I. Figure 23 depicts the effects of three planting dates on flowering times at Dalby, Darling Downs, Queensland. Table 3 shows preferred planting times for various regions in Queensland and NSW.

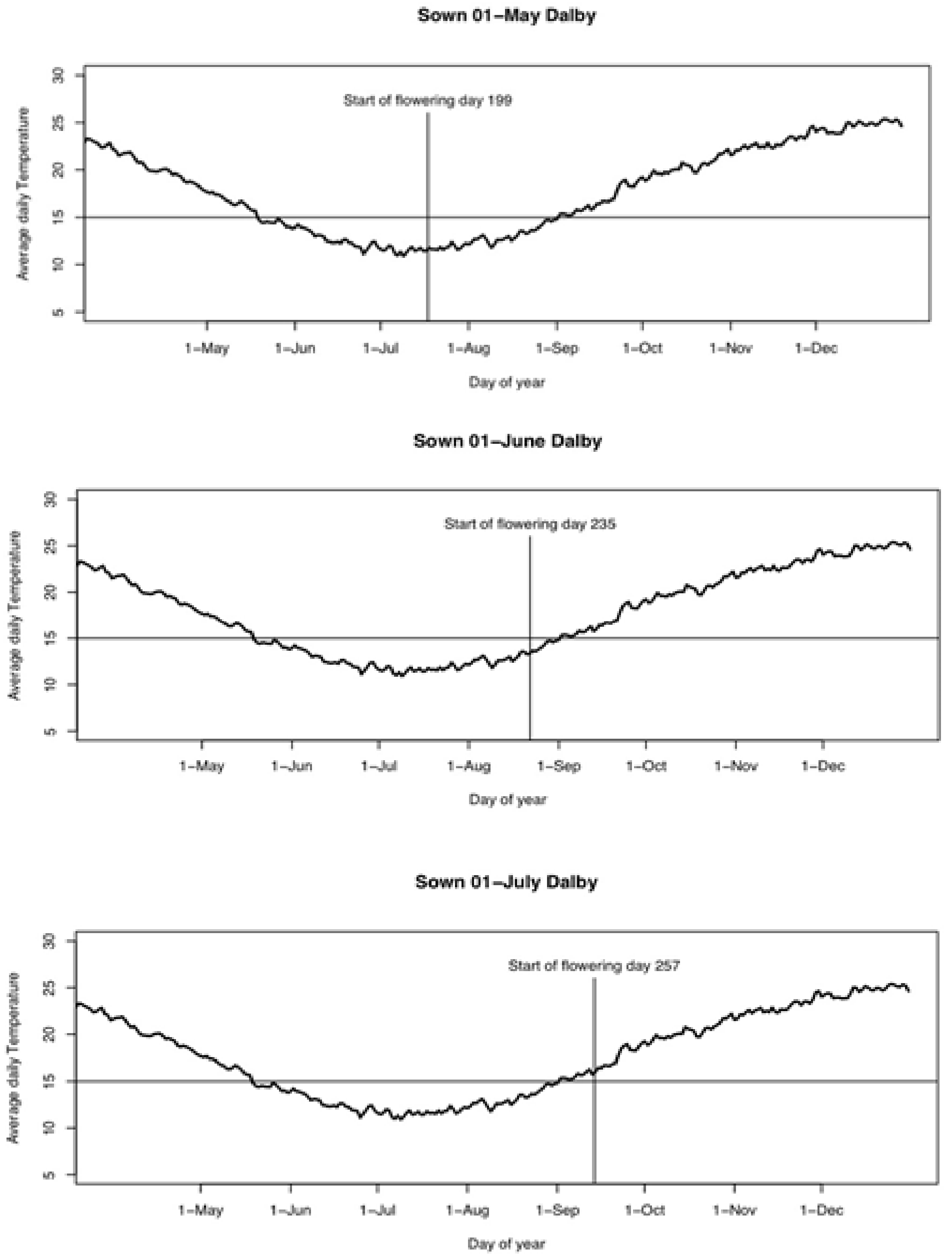
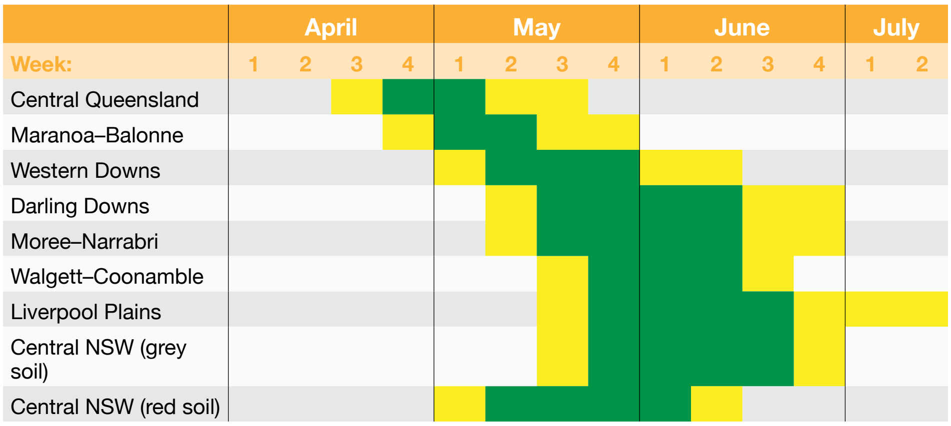


Figure 23: Effect of planting date on flowering in the Dalby Region.

Table 3: Preferred planting times for different regions



Yellow boxes indicate marginal sowing time, where increased costs and/or lower yields are likely; green boxes indicate preferred sowing window

4.4 Reliable chickpea yields: risk management

4.4.1 Soil water storage capacity

Calculation or estimation of a yield expectation for chickpeas requires knowledge of the plant-available water-holding capacity (PAWC) for a soil type and of how full this capacity is.

Table 4 shows the approximate PAWC for a range of soil types with original vegetation. The table does not consider factors within the soil such as sodic layers, compaction or salinity that may be present in some areas.

Table 4: Plant-available soil water (mm) for a range of soils

Soil type	Total plant-available soil water	0–30 cm	30–60 cm	60–90 cm	90–120 cm
Heavy alluvial (e.g. silty clay)	250	70	70	60	50
Heavy black earth (e.g. Waco)	220	70	70	50	30
Less heavy black earth	190	60	60	40	30
Heavy box	160	50	40	40	30
Uplands Brigalow	140-150	40	40	30	30
Grey clay (e.g. Coolibah)	150	40	40	30	30
Open Downs	130-150	40	40	30	20
Red earth (Western Downs)	110	40	30	30	10
Red Ferrosol (krasnozem)	100	30	30	30	10
Shallow clay (Central Highlands)	70-90	40	30	–	–

Note: Values are approximations only and individual soils will vary around the value quoted

4.4.2 Rooting depth

Chickpeas can access moisture to 90 cm depth provided there is no compaction or saline/sodic layers in the soil profile.

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. [12]

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

4.5 Yield expectation: relation to starting soil water and location

Starting soil water can have a strong influence on the yield expectation of chickpea as well as the riskiness of production. Table 5 presents the average and range of yields in good and poor years.

The values in the table are derived from the simulation model APSIM, which has been tested in the northern region over the past 10 years. Values are APSIM-simulated chickpea yields for three conditions of starting plant-available water at eight locations in the northern grains region. Simulations were conducted with 100 years of daily historical climate data at each location. The simulation setup involved cv. Amethyst sown in late May at 20 plants/m^2. The values are a conservative estimate. [13]

Table 5: Potential chickpea grain yield (kg/ha) in a range of years with different starting soil water levels

Location	Starting soil water (mm)	Driest 10%of years	Driest 25%of years	Average	Wettest 25% of years	Wettest 10% of years
Biloela	170	897	1065	1707	2318	3022
	100	355	413	853	1201	1828
	66	259	337	767	1047	1782
Emerald	170	967	1231	1742	2278	2850
	100	347	442	854	1093	1842
	66	240	354	754	922	1749
Dalby	170	1164	1609	2202	2867	3180
	100	763	1093	1819	2660	3000
	66	344	598	1260	1895	2635
Roma	170	885	1056	1587	2083	2556
	100	462	571	1062	1374	1891
	66	319	417	898	1206	1833
Moree	170	1155	1449	2026	2584	2924
	100	749	931	1686	2325	2856
	66	297	453	1142	1667	2416
Kingaroy	170	1181	1670	2354	3071	3378
	100	577	803	1449	2032	2643
	66	460	649	1288	1820	2424
Walgett	170	981	1359	1840	2399	2741
	100	627	844	1409	1920	2561
	66	242	367	814	1007	1880
Goondiwindi	170	1065	1356	1884	2446	2830
	100	641	904	1505	1934	2718
	66	282	439	975	1349	2309

(Source: M. Robertson CSIRO.)

4.6 Seasonal climate outlook and yield expectation

The Southern Oscillation Index (SOI) can be a strong predictor of the likelihood of an above- or below-average seasonal climate outlook, particularly for winter cropping in the northern grains region. Knowledge of pre-season SOI and models estimates of yield expectations can be used to provide yield probabilities.

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

Table 6 shows the influence of SOI phase in May preceding the winter season on average simulated chickpea yields at three locations in the northern grains region. The simulations show that a consistently negative SOI in May will predict a significantly lower yield expectation than all other cases of SOI. The values are derived from the simulation model APSIM. [14]

Table 6: Effect of Southern Oscillation Index (SOI) on chickpea grain yield (kg/ha) with a range of starting soil water

The yields in the 'All years' column are average yields derived from Table 5

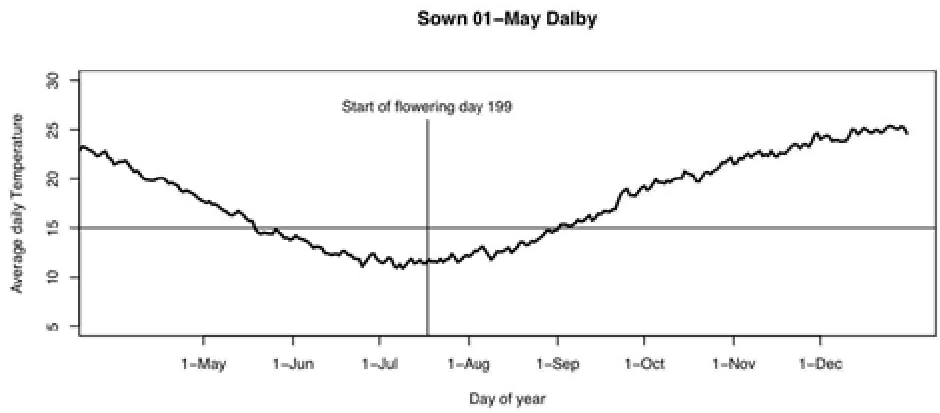
Location	Starting soil water (mm)	SOI					
		All years	Falling	Negative	Positive	Rising	Zero
Emerald	170	1742	1761	1428	1817	1836	1741
	100	854	849	600	923	929	857
	66	754	733	498	798	880	738
Dalby	170	2202	2006	1769	2262	2347	2355
	100	1819	1551	1324	1862	1982	2040
	66	1260	953	824	1265	1426	1498
Roma	170	1587	1442	1270	1659	1691	1673
	100	1062	941	774	1116	1164	1135
	66	898	755	612	963	996	977
Moree	170	2026	1910	1780	2177	2204	1924
	100	1686	1531	1350	1877	1890	1595
	66	1142	952	874	1386	1276	1047
Walgett	170	1840	1666	1539	1927	2031	1815
	100	1409	1186	1078	1536	1599	1389
	66	814	601	619	934	921	812
Goondiwindi	170	1884	1772	1521	1941	2019	1989
	100	1505	1433	1120	1549	1602	1646
	66	975	917	618	997	1042	1136

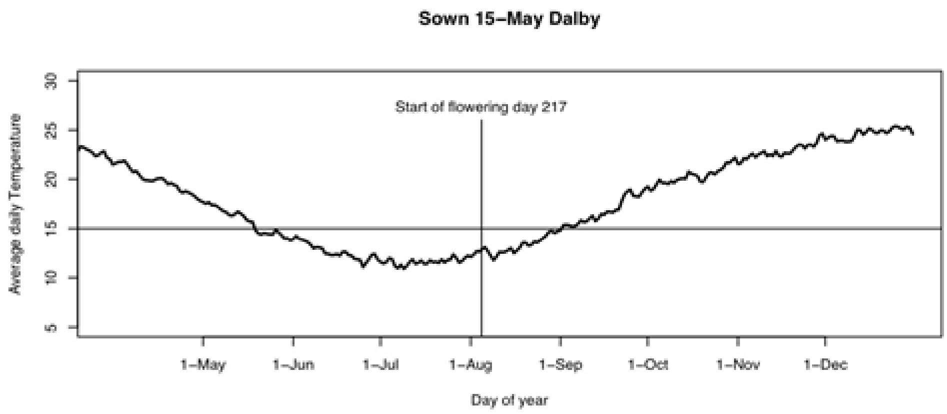
(Source: M. Robertson, CSIRO.)

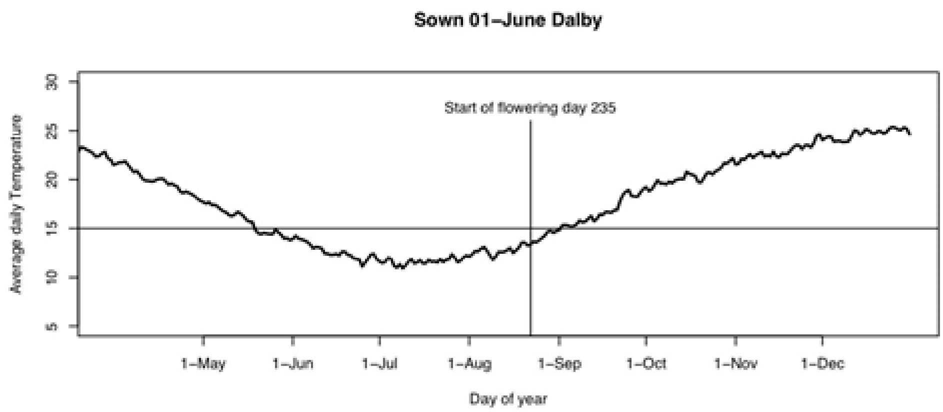
[14] Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

4.7 Appendix I. Effect of planting time on flowering [15]

4.7.1 Effect of planting time on flowering in the Dalby region

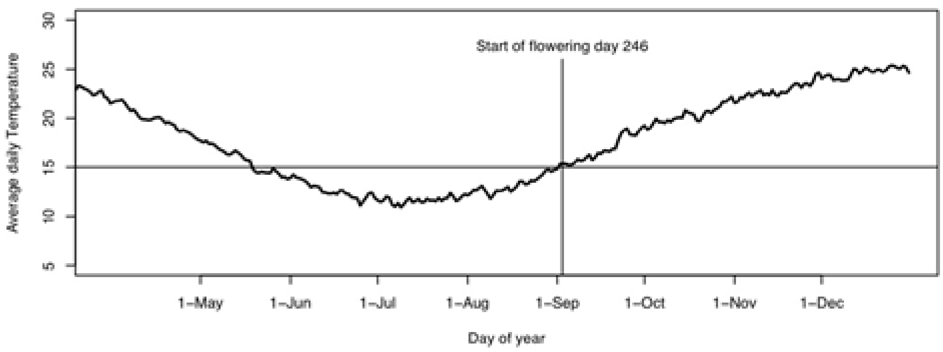




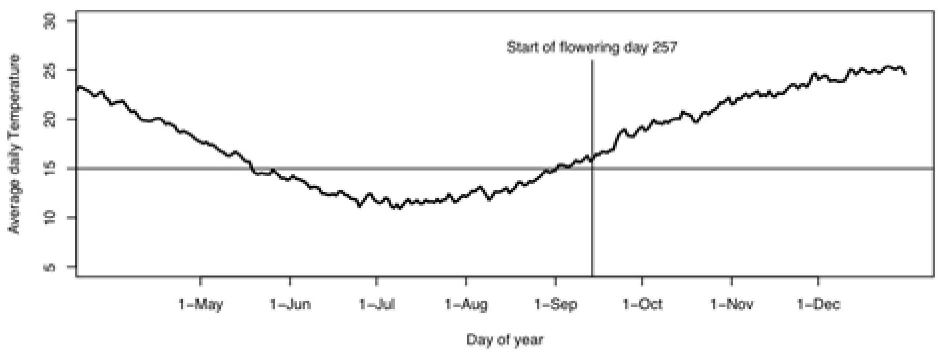


15 Pulse Australia (2013) Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

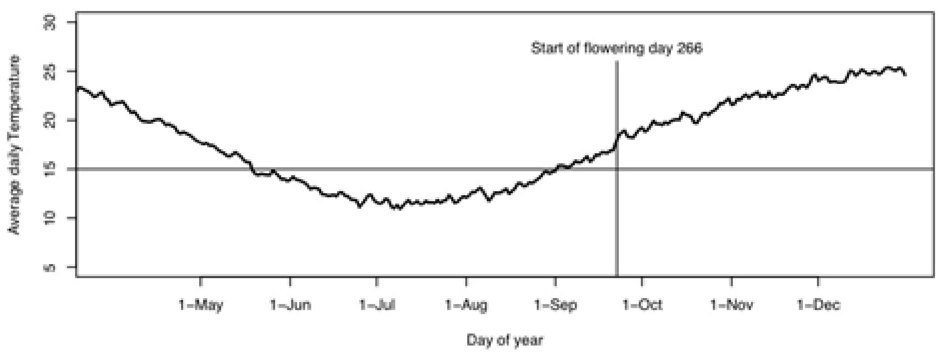
Sown 15–June Dalby



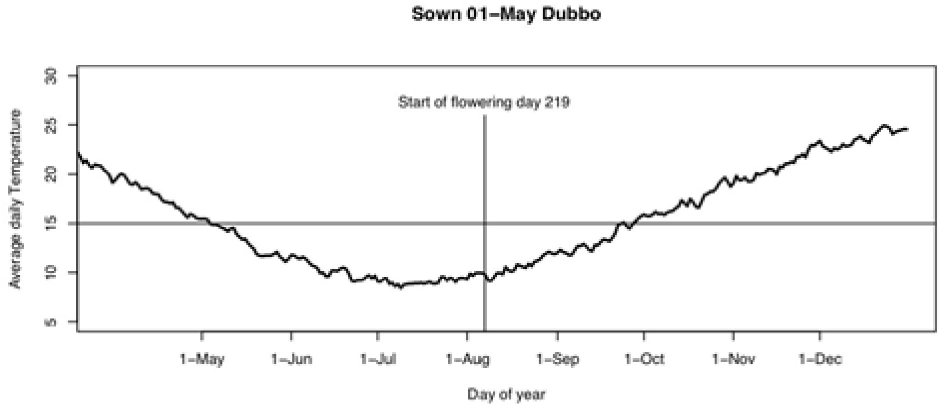
Sown 01–July Dalby

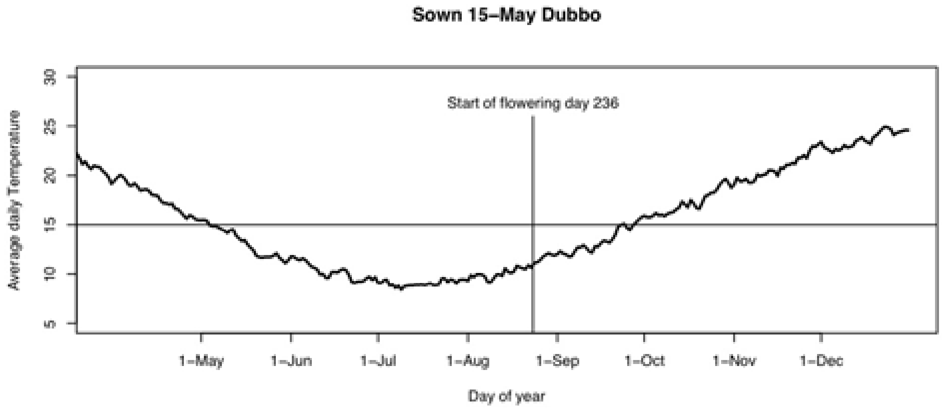


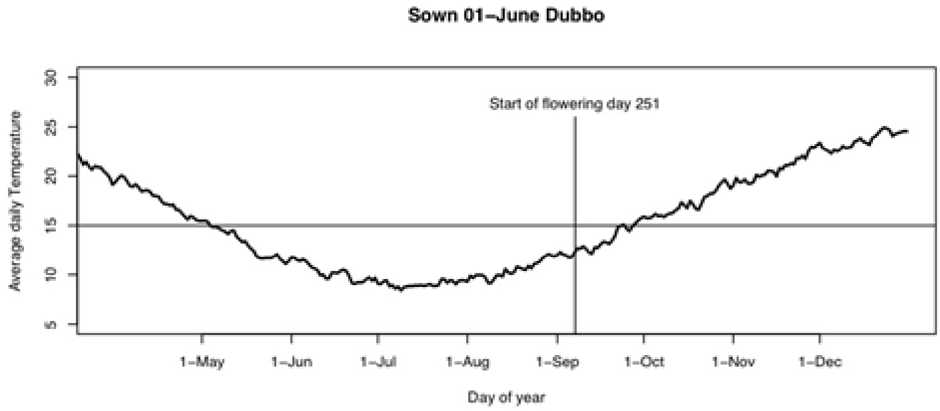
Sown 15–July Dalby

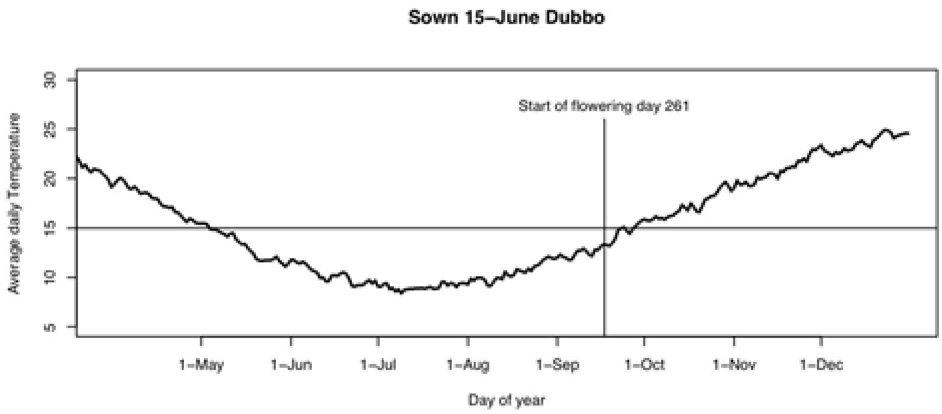


4.7.2 Effect of planting time on flowering in the Dubbo region

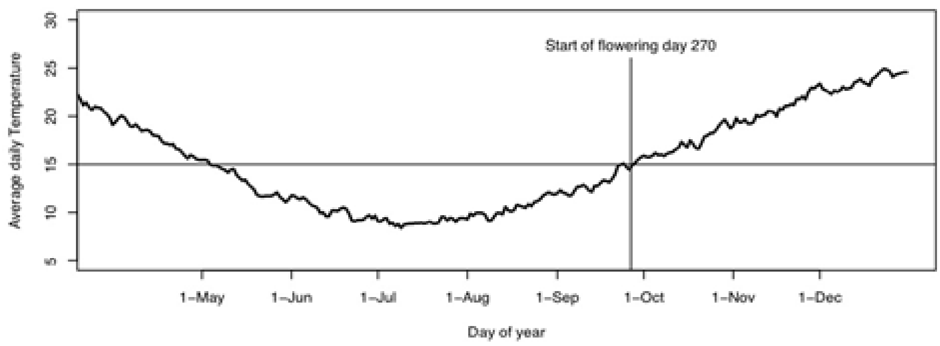




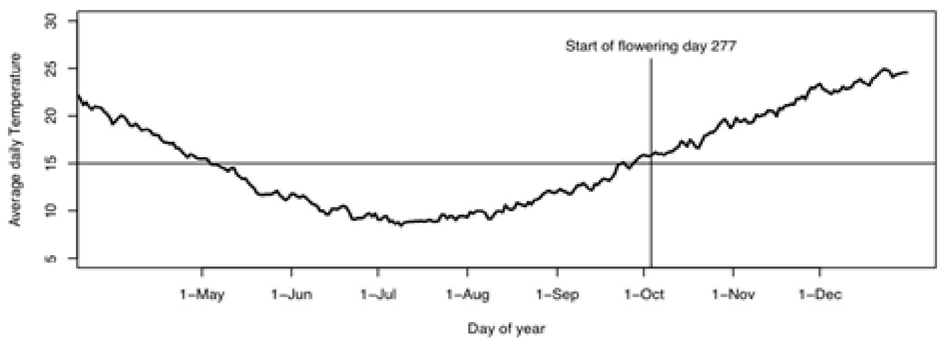




Sown 01–July Dubbo

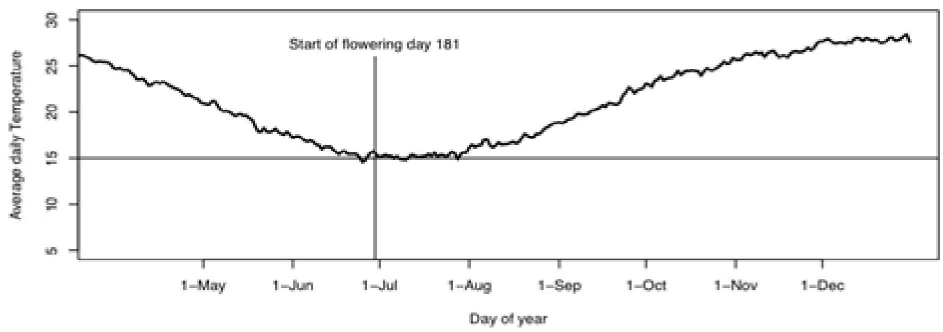


Sown 15–July Dubbo

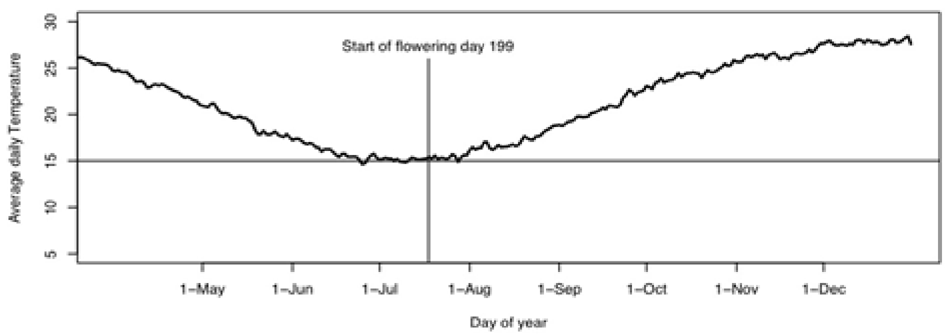


4.7.3 Effect of planting time on flowering in the Emerald region

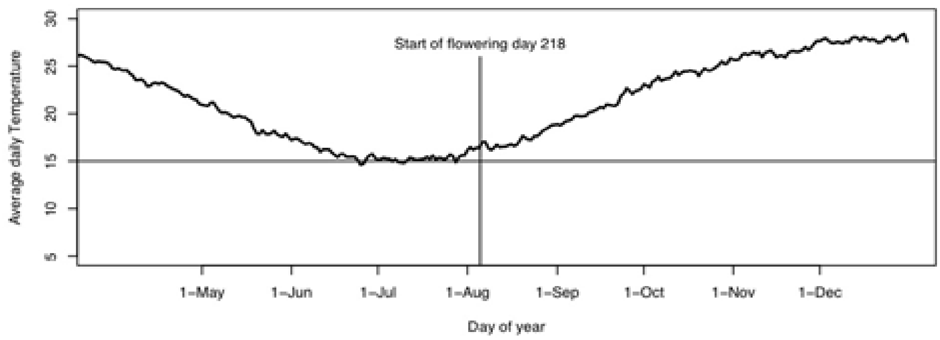
Sown 01–May Emerald



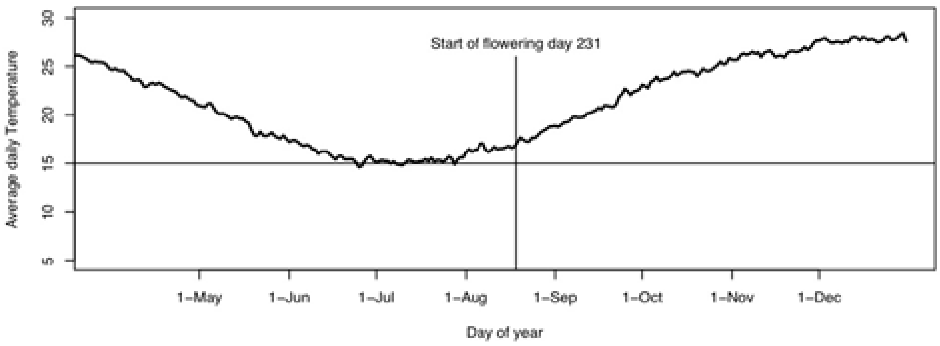
Sown 15–May Emerald



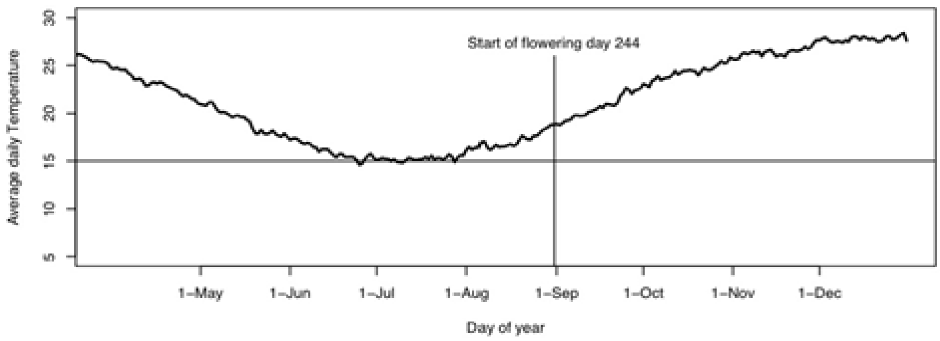
Sown 01–June Emerald



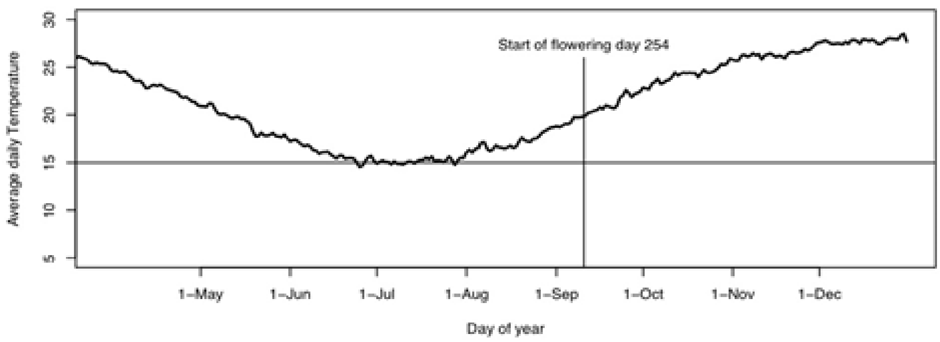
Sown 15–June Emerald



Sown 01–July Emerald

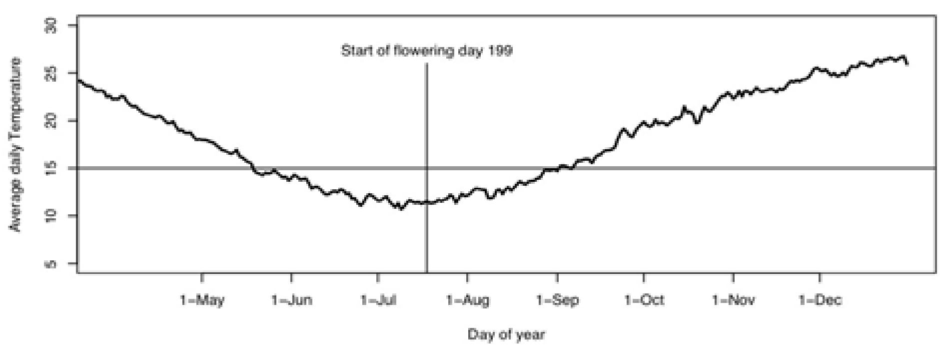


Sown 15–July Emerald

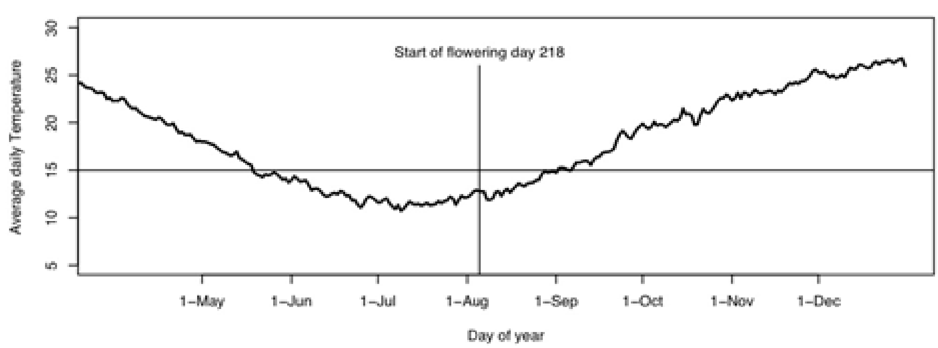


4.7.4 Effect of planting time on flowering in the Goondiwindi region

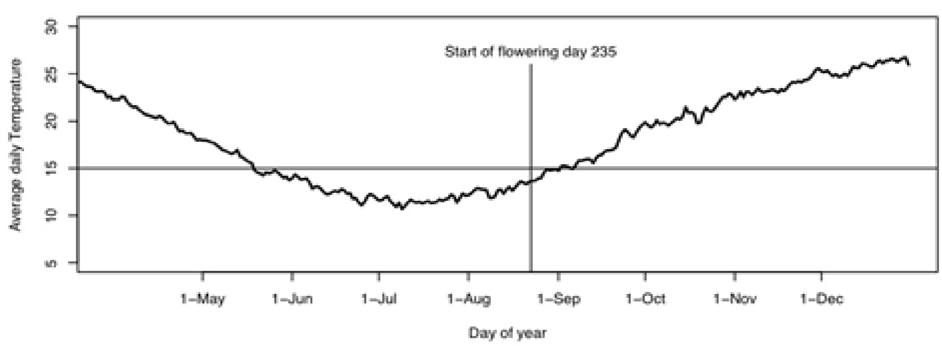
Sown 01–May Goondiwindi



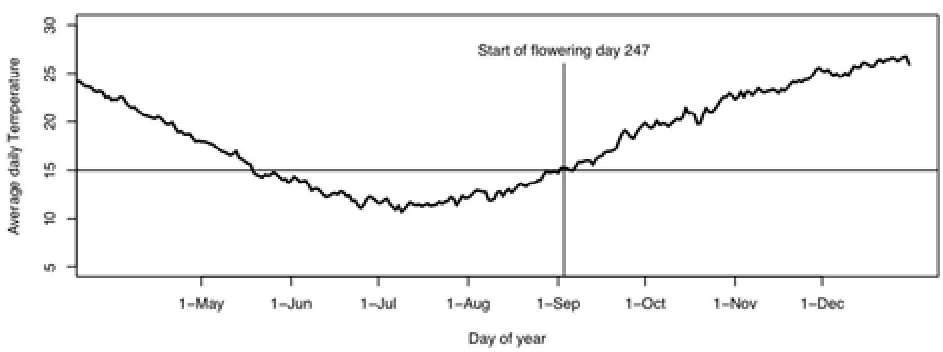
Sown 15–May Goondiwindi



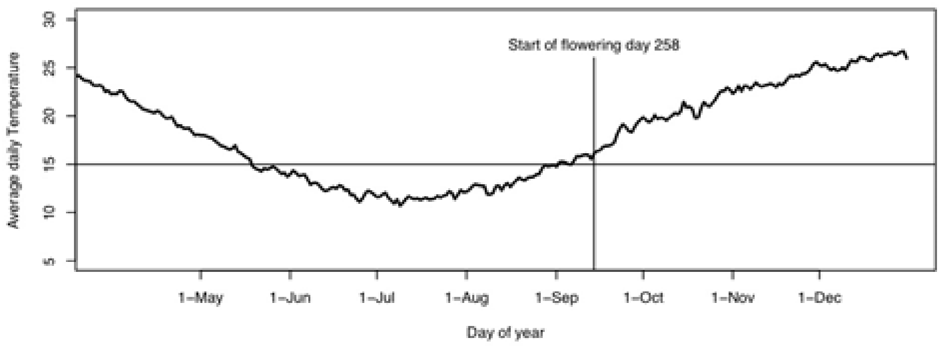
Sown 01–June Goondiwindi



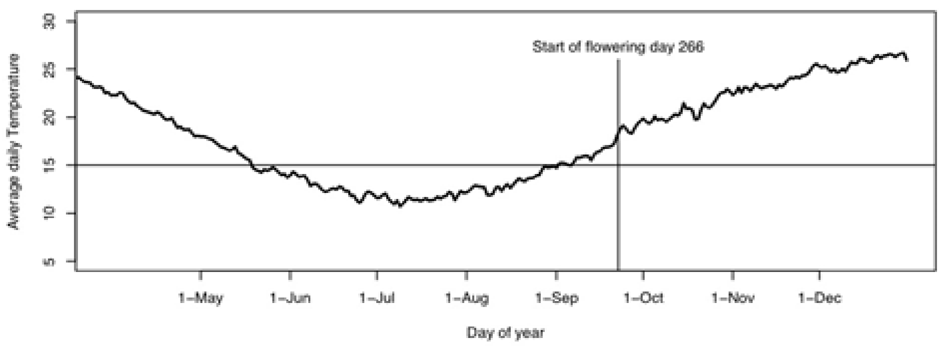
Sown 15–June Goondiwindi



Sown 01–July Goondiwindi

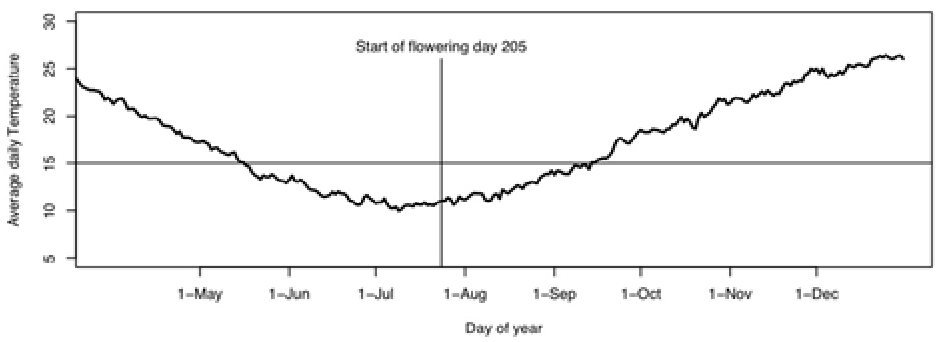


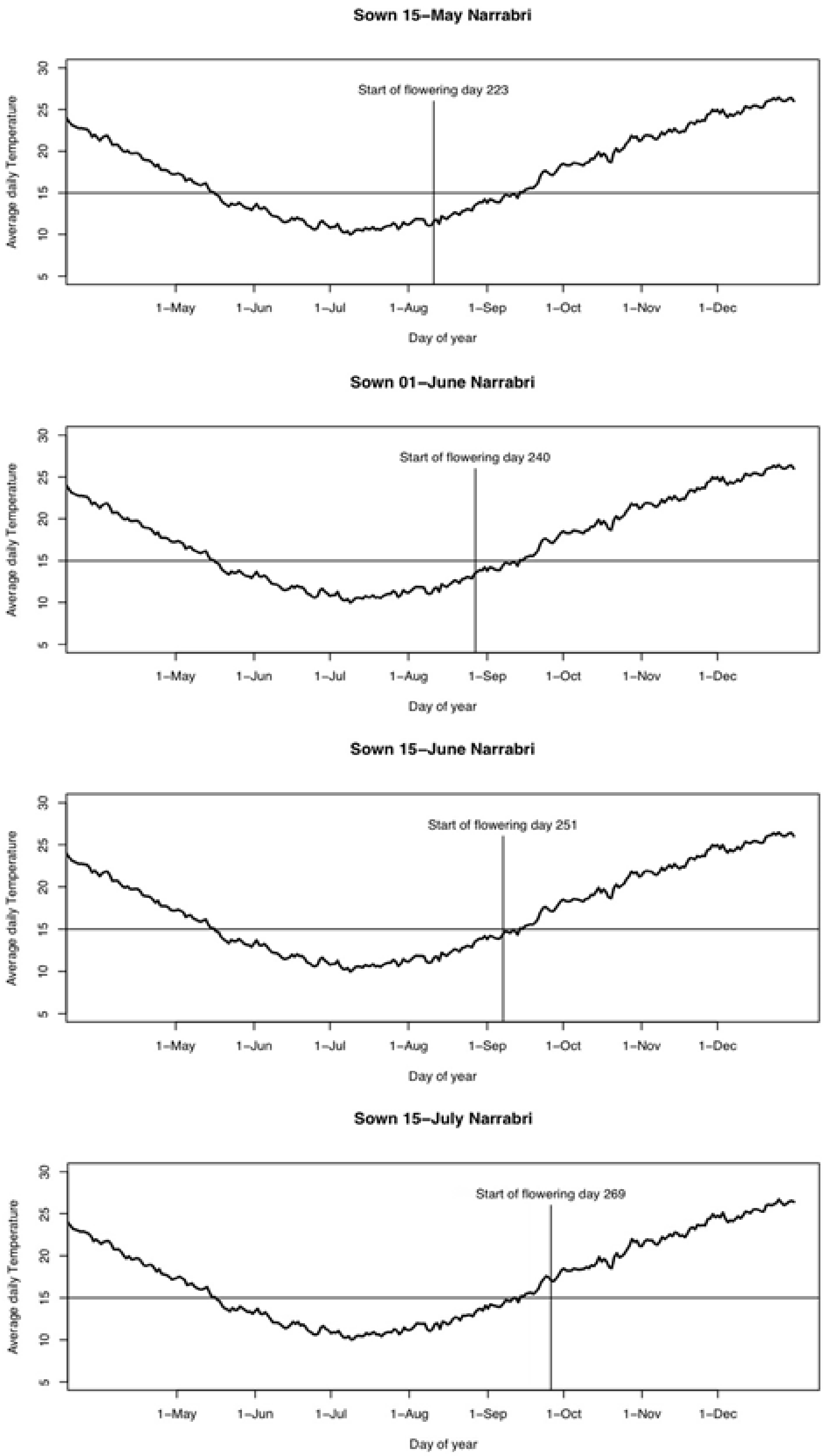
Sown 15–July Goondiwindi



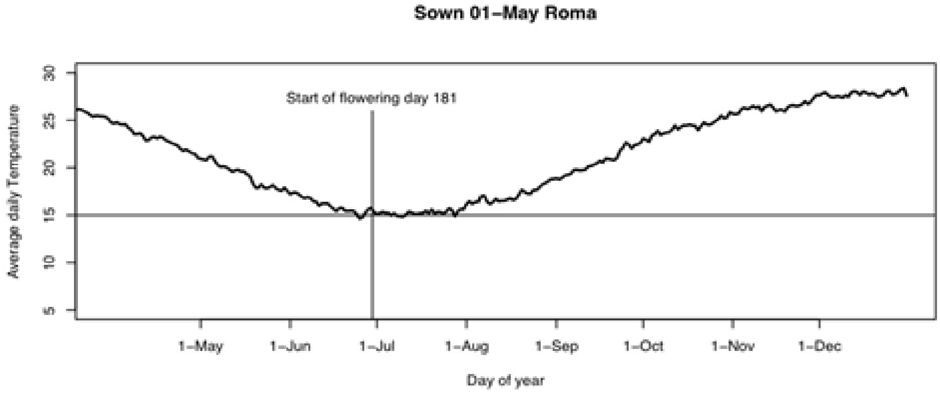
4.7.5 Effect of planting time on flowering in the Narrabri region

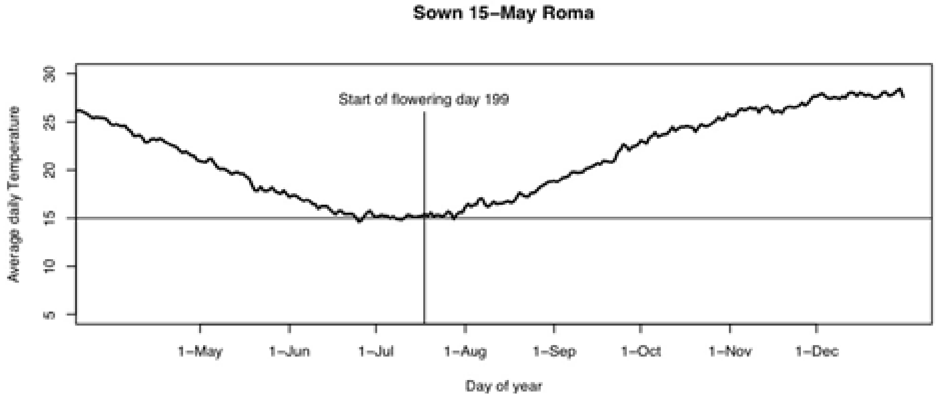
Sown 01–May Narrabri

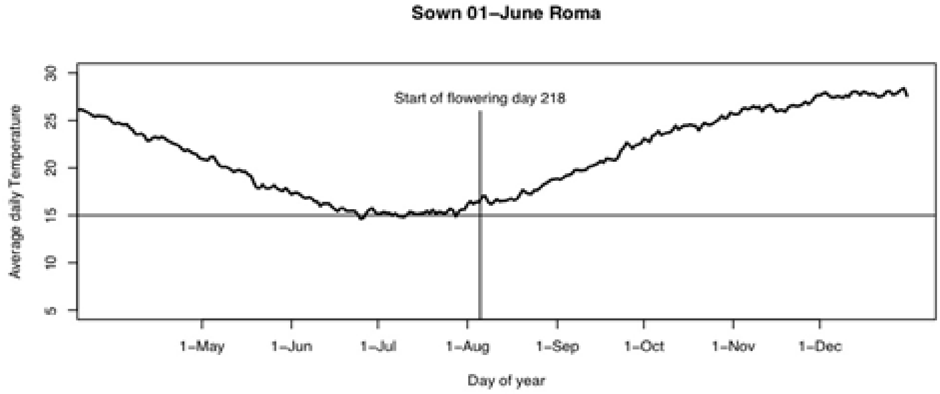


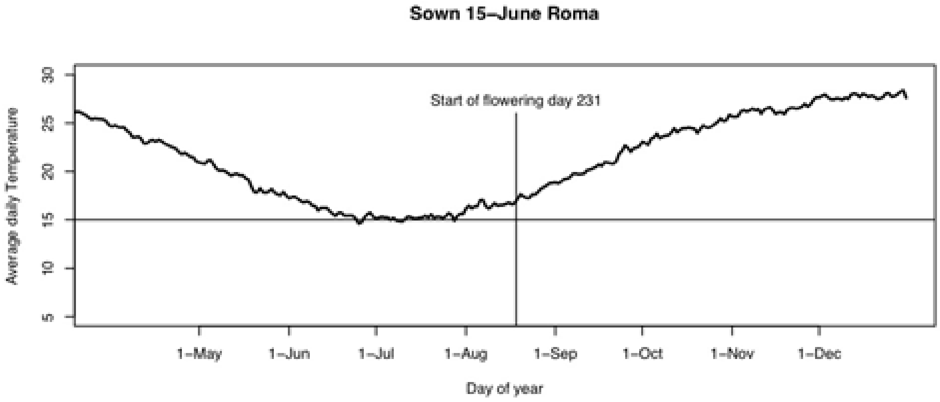

Sown 15–May Narrabri
Start of flowering day 223
Sown 01–June Narrabri
Start of flowering day 240
Sown 15–June Narrabri
Start of flowering day 251
Sown 15–July Narrabri
Start of flowering day 269
Average daily Temperature
5
10
15
20
25
30
1–May
1–Jun
1–Jul
1–Aug
1–Sep
1–Oct
1–Nov
1–Dec
Day of year

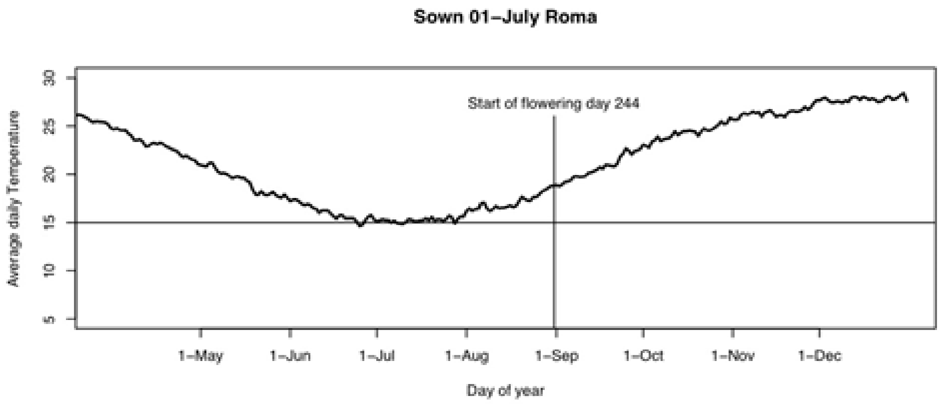
4.7.6 Effect of planting time on flowering in the Roma region

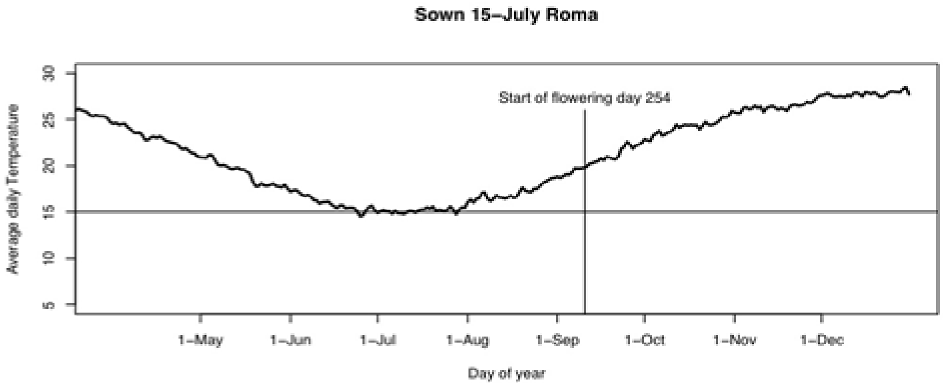




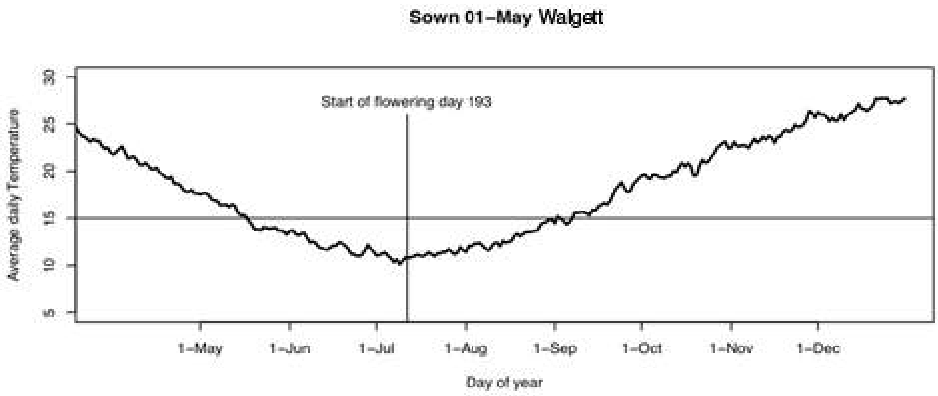


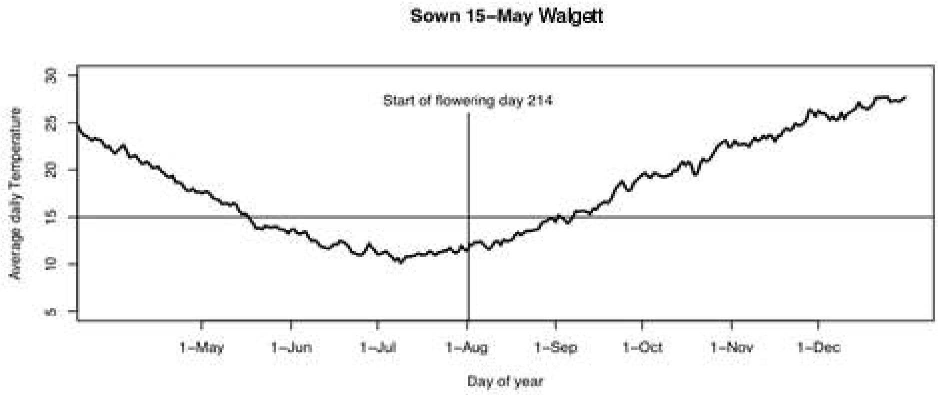


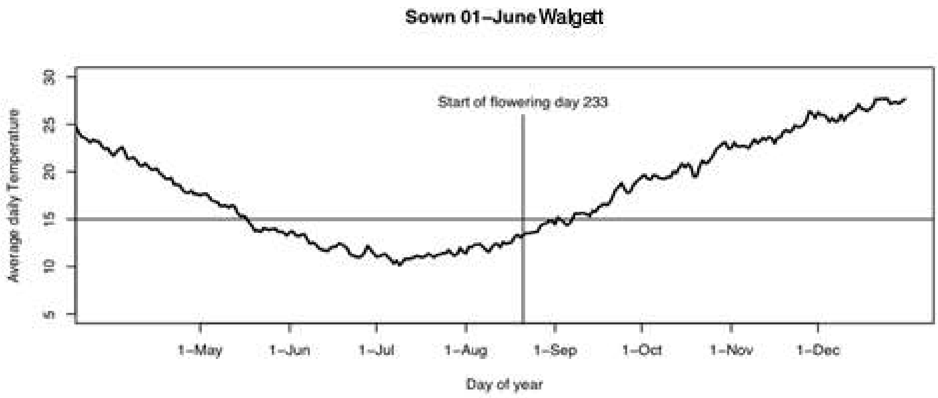


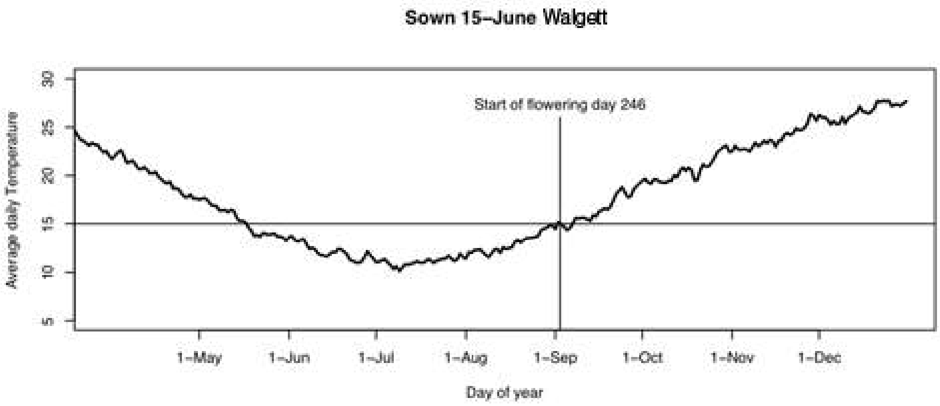


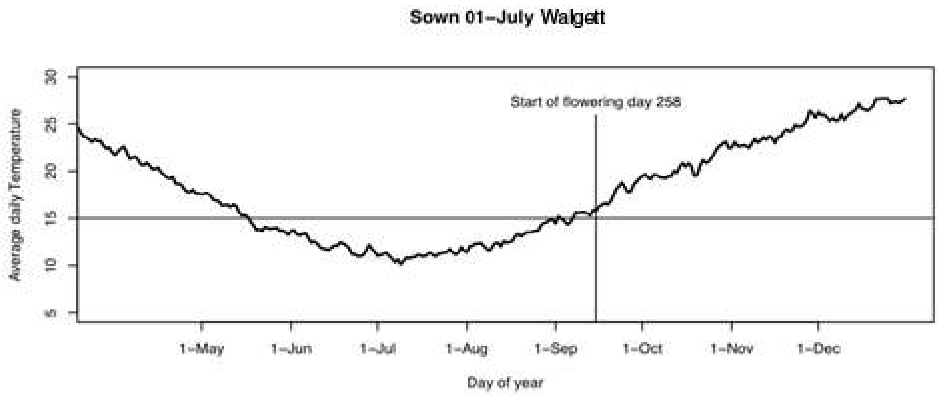
4.7.7 Effect of planting time on flowering in the Walgett region

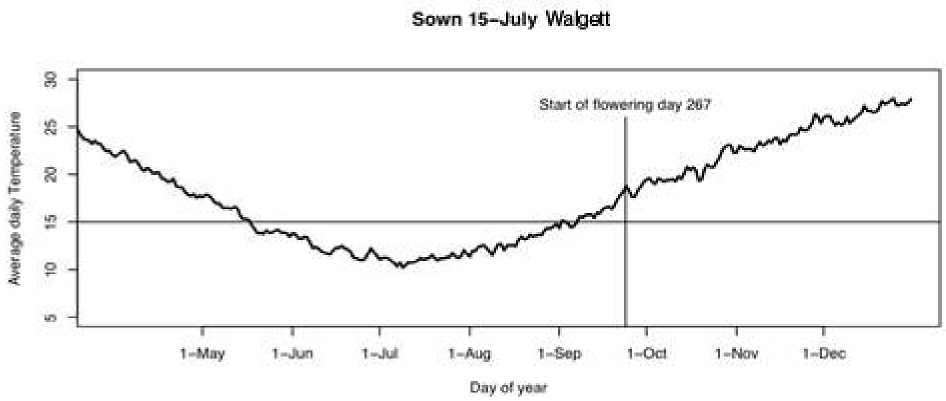



Sown 15–May Walgett
Start of flowering day 214
Average daily Temperature
5
10
15
20
25
30
1–May
1–Jun
1–Jul
1–Aug
1–Sep
1–Oct
1–Nov
1–Dec
Day of year


Sown 01–June Walgett
Start of flowering day 233
Average daily Temperature
5
10
15
20
25
30
1–May
1–Jun
1–Jul
1–Aug
1–Sep
1–Oct
1–Nov
1–Dec
Day of year


Sown 15–June Walgett
Start of flowering day 246
Average daily Temperature
5
10
15
20
25
30
1–May
1–Jun
1–Jul
1–Aug
1–Sep
1–Oct
1–Nov
1–Dec
Day of year


Sown 01–July Walgett
Start of flowering day 258
Average daily Temperature
5
10
15
20
25
30
1–May
1–Jun
1–Jul
1–Aug
1–Sep
1–Oct
1–Nov
1–Dec
Day of year



4.8 References and further reading

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F Singh, B Diwakar (1995) 'Chickpea botany and production practices.' ICRISAT Skills development Series No 16, www.icrisat.org/what-we-do/learning-opportunities/lsu-pdfs/sds.16.pdf

J Whish, B Cocks (2011) Sowing date and other factors that impact on pod-set and yield in chickpea. GRDC Update paper Goondiwindi, Qld, www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2011/04/Sowing-date-and-other-factors-that-impact-on-podset-and-yield-in-chickpea

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A Srinivasan, C Johansen, NP Saxena (1997) 'Cold tolerance during early reproductive growth of chickpea *(Cicer arietinum L.): characterization of stress and genetic variation in pod set. Field Crops Research* 57(2), 181-193, http://oar.icrisat.org/1716/

http://www.apsim.info/Documentation/Model,CropandSoil/CropModuleDocumentation/Chickpea.aspx

SECTION 5

Nutrition and fertiliser

More information

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/02/Virus-in-chickpea-in-northern-NSW-2012

Incorrect levels of nutrients (too little, too much or the wrong proportion) can cause nutritional problems. 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]

Plant tissue analysis can play an important role in detecting non-visible or subclinical symptoms, and in fine-tuning nutrient requirements. This is particularly helpful where growers are aiming to capitalise on available moisture.

Tissue tests also help to identify the cause of symptoms being expressed by plants but not fitting a visual diagnosis. Technology is enabling quicker analysis and reporting of results to enable foliar or soil-applied remedies to be applied in a timely manner for a quick crop response. [3]

More information

http://www.grdc.com.au/GRDC-FS-NFixation-Chickpeas

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Changing-nutrient-management-strategies-in-response-to-declining-background-fertility

http://www.grdc.com.au/uploads/documents/4%20Nutrition.pdf

5.1 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

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

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

More information

http://www.grdc.com.au/GRDC-FS-BFDCN

J Paterson (2014). Deep soil tests reveal northern nutrient deficiencies

like deficiency of another. For instance, molybdenum (Mo) is required by pulses to complete the process of nitrogen (N) fixation. [4]

Recent research by Dr Mike Bell, Principal Research Fellow at the University of Queensland's Queensland Alliance for Agriculture and Food Innovation (QAAFI), shows that many farms in central Queensland have phosphorus (P) and potassium (K) concentrated in the topsoil and critically low levels in the subsoil. Plants cannot access these immobile nutrients when the topsoil is dry, and this reduces productivity. [5]

5.1.1 Soil testing

In northern cropping soils, nutrient deficiencies other than N are a relatively recent development. Consequently, limited nutrient research has been conducted in these soils and in the many crop types grown in northern cropping systems. Most research has been done in wheat and barley.

More information

http://www.grdc.com.au/Research-and-Development/Major-Initiatives/More-Profit-from-Crop-Nutrition

http://www.publish.csiro.au/pid/5352.htm

Recent research has highlighted that N applications can be wasted, even on cropping soils that have low N availability, if the levels of other nutrients such as K, P and sulfur (S) are not adequate. The importance of subsoil layers for nutrients such as P and K is not yet reflected in the limited soil test–crop response data available.

Researchers are using rough rules-of-thumb to help interpret P and K soil tests in terms of likely fertiliser responsiveness on northern region Vertosols.

5.1.2 Types of soil test

Appropriate soil tests for measuring soil extractable or plant-available nutrients in the northern cropping region are:

- bicarbonate-extractable P (Colwell-P), to assess easily available soil P
- acid-extractable P (BSES-P), to assess slower release soil P reserves and the build-up of fertiliser residues (not required annually)
- exchangeable K
- KCl-40-extractable S or MCP-S
- 2 m KCl-extractable mineral N, to provide measurement of nitrate-N and ammonium-N

The more attention we pay to all of the activities that contribute to nutrient management (Figure 1), the better the outcome we will get from soil and plant testing. Testing may not provide a useful contribution if one or more of these steps is not done well. [6]

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

5 N Baxter (2013) Trials measure chickpea/rotation profit. GRDC Ground Cover Issue 107, Nov.–Dec. 2013, http://grdc.com.au/Media-Centre/Ground-Cover/Ground-Cover-Issue-107-NovDec-2013/Trials-measure-chickpea-wheat-rotation-profit

6 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

More information

http://grdc.com.au/Resources/Factsheets/2013/11/Better-fertiliser-decisions-for-crop-nutrition

www.grdc.com.au/GRDC-FS-SoilTestingN

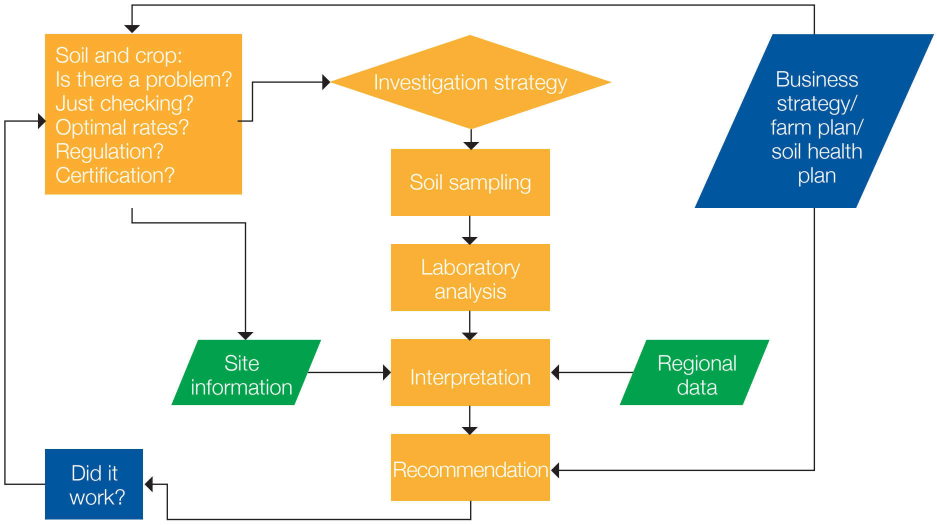

Figure 1: Nutrient management flow chart.

5.2 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 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 useful. 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 increase and may restrict plant growth, usually by restricting the rhizobia and so 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. [7]

5.3 Balancing inputs

If the nutrients (P, N, Zn, etc.) removed as grain from the paddock are not replaced, then crop yields and soil fertility 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 about 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. [8]

More information

http://www.publish.csiro.au/paper/AR9931403

http://bign.com.au/Latest%20News/Perfect%20Pulses

http://www.publish.csiro.au/paper/EA9860353

http://www.publish.csiro.au/?paper=EA98027

http://www.publish.csiro.au/paper/EA97078.htm

http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0009/166329/phosphorous-winter-crops.pdf

http://www.incitecpivotfertilisers.com.au/News/Latest%20News/Perfect%20Pulses

Table 1: Amounts of macro- and micro-nutrients removed per tonne of grain

Note: These values should be used as a guide only

Grain	N	P	K	S	Ca	Mg	Cu	Zn	Mn
			(kg)					(g)	
Pulses									
Chickpea (Desi)	33	3.2	9	2.0	1.6	1.4	7	34	34
Chickpea (Kabuli)	36	3.4	9	2.0	1.0	1.2	8	33	22
Faba bean	41	4.0	10	1.5	1.3	1.2	10	28	30
Lentil	40	3.9	8	1.8	0.7	0.9	7	28	14
Lupin (sweet)	53	3.0	8	2.3	2.2	1.6	5	35	18
Lupin (white)	60	3.6	10	2.4	2.0	1.4	5	30	60
Field pea	38	3.4	9	1.8	0.9	1.3	5	35	14
Cereals									
Wheat	23	3.0	4	1.5	0.4	1.2	5	20	40
Barley	20	2.7	5	1.5	0.3	1.1	3	14	11
Oats	17	3.0	5	1.6	0.5	1.1	3	17	40

Source: Grain Legume Handbook.

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. [9]

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.

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

5.4 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.

Table 2: An example of nutrient budgeting

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, 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 in northern NSW.

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.

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. [10]

Table 3: Fertiliser application rate ready-reckoner (all rates are kg/ha) for some of the fertilisers used on pulses

P	Superphosphate						6 : 16 : 0 : 10 Legume Special			10 : 22 : 0 MAP		18 : 20 : 0 DAP		0 : 15 : 0 : 7 Grain Legume Super	
	Single 8.6% P		Gold Phos 10 18% P		Triple 20% P										
	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

5.4.1 Detecting nutrient deficiencies

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).

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.

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

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	5 μg/g (ppm)		
Adequate	8 μg/g		

Source: Grain Legume Handbook, 2008.

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.

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 5 lists the plant analysis criteria for chickpea. These should be used as a guide only. Care should be taken to use plant tissue tests for the intended purpose. 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. [11]

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 5) 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.

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

Table 5: 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

5.4.2 Nutrient toxicity

Soil pH affects the availability of most nutrients. Occasionally, some nutrients are present in the soil 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 (Table 6).

Boron (B) toxicity occurs on many of the alkaline soils of the southern cropping areas (Figure 2; see also Mn toxicity in Figure 3). The most characteristic symptom of B toxicity in pulses is chlorosis (yellowing), and if severe, some necrosis (death) of leaf tips or margins. Older leaves are usually more affected. There appears to be little difference in reaction between current varieties of chickpeas. [12]

Table 6: Pulse reactions to nutrient toxicities

	Boron	Aluminium	Manganese
Chickpea	Sensitive	Very sensitive	Very sensitive
Faba bean	Tolerant	Sensitive	Sensitive
Lentil[A]	Very sensitive	Very sensitive	Very sensitive
Lupin[A]	Not grown	Tolerant	Tolerant
Field pea	Sensitive	Sensitive	Sensitive

[A]Not usually grown on alkaline–high boron soils.

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

Figure 2: Symptoms of boron toxicity in chickpea leaves. (Source: CSIRO)

Figure 3: Symptoms of manganese toxicity in chickpea leaves. (Source: CSIRO)

5.5 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.

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. Neutralized 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

More information

http://www.daf.qld.gov.au/plants/field-crops-and-pastures/broadacre-field-crops/chickpeas/nutrition

Neutral fertilisers include:

- 'Super lime'

Alkaline fertilisers include:

- DAP also known as 18 : 20 : 0
- starter NP
- lime [13]

5.5.1 Pulses and 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.

Deep banding of fertiliser is preferred for some pulses, 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. [14]

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.

5.5.2 Nitrogen

If chickpea plants have effectively nodulated, they should not normally need N fertiliser.

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 [15]

Symptoms of N deficiency are depicted in Figure 4, and an N balance for chickpeas is presented in Table 7. Chickpea grain contains 3234 kg N/t. [16]

Figure 4: Nitrogen deficiency in chickpeas. Plants show signs of stunting, yellowing and poor growth. (Source: CSIRO)

More information

M Bell, G Schwenke, D Lester (2016), Understanding and managing N loss pathways

B O'Mara, C Walker (2015), Lessons learnt about nitrogen and phosphorus from a 30 year study in a sub-tropical continuous cropping system on a vertosol

Figure 5: Nitrogen balance for chickpeas

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

Grain harvest index (HI) is the grain yield as a percentage of total shoot dry matter production (average about 40%)

More information

http://www.publish.csiro.au/paper/SR9960273.htm

http://www.grdc.com.au/uploads/documents/dnr00004.pdf

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

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

More information

[D Lester, M Bell, R Graham, D Sands, G Brooke (2016), Phosphorus and potassium nutrition]

[K Andersson, M Tighe, C Guppy, P Milham, T McLaren (2016), The mobility of P in alkaline clays of the Northern Grains Region]

[A Verrell, L Jenkins (2016), Nutrition in chickpea 2015 (northern NSW pulse agronomy project)]

[M Bell, D Sands, D Lester, R Norton (2015), Response to deep placed P, K and S in central Queensland]

[A Verrell, L Jenkins (2015), Effect of macro and micro nutrients on grain yield in chickpea crops at Trangie and Coonamble]

5.5.3 Phosphorus

Chickpeas are adapted to alkaline soils with high levels of unavailable P, and they have evolved methods of extracting P from the soil that would be inaccessible to many other pulse and cereal crops (see Figure 5 for symptoms of P deficiency).

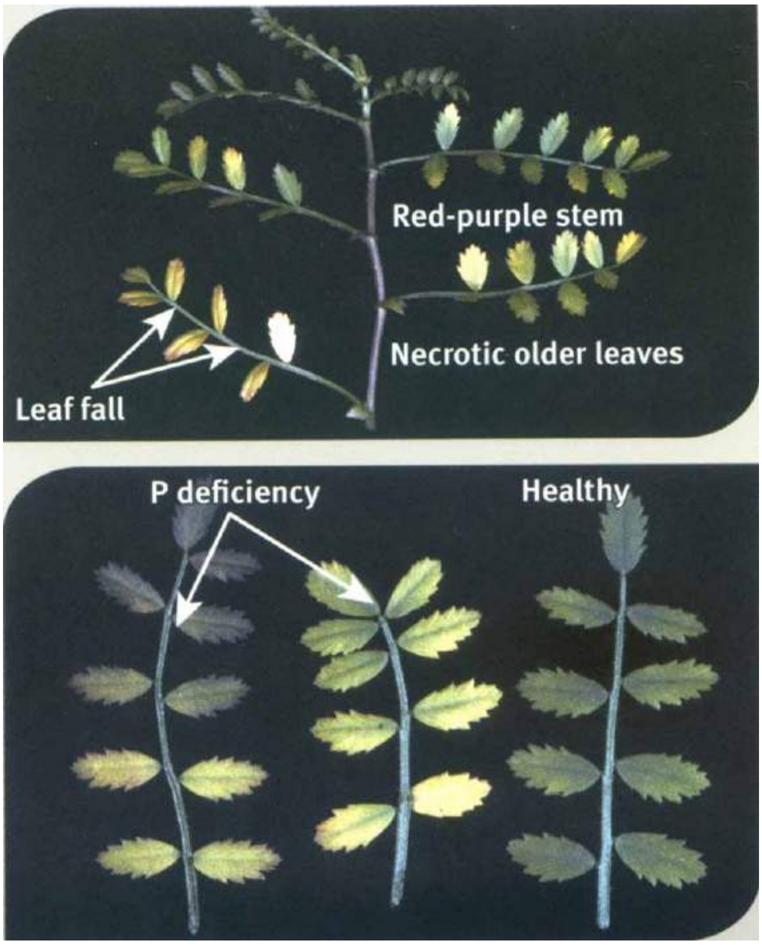

Figure 6: Phosphorus deficiency symptoms in chickpea leaves. (Source: CSIRO)

This ability is largely due to a combination of two factors. First, chickpeas secrete strong organic acids from their roots. These acids dissolve insoluble forms of P in the soil and convert them to water soluble P, which are then available for plant uptake.

Second, AMF colonise and build up to very high levels on the chickpea root system. The fungi produce hyphae that colonise the root and then grow out into the soil (much further than root hairs). Phosphorus and Zn are taken up by the hyphae and transported back for use by the chickpea plant. AMF build up to much greater levels on chickpeas than on wheat root systems (up to five times higher levels).

Although chickpeas are considered to have moderately high crop requirement for P, economic fertiliser responses to P are uncommon.

Most of the research from Australia and overseas indicates that uptake of P is far more efficient in chickpeas than in other winter crops, and responses are usually small and uneconomic.

Possible exceptions are:

- soils with critically low P levels (<6 mg P/kg) and no prior history of P fertiliser; and
- long fallow situations with low AMF levels (≥10 months), where later plantings into cold wet soils can limit or slow the root system development.

Because P is immobile in the soil, these situations can induce P deficiency.

High AMF situations

Where soil AMF levels are moderate–high (double-crop situations or short, 6-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 8), 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 9), this may not always translate into a positive yield response.

Department of Agriculture Fisheries and Forestry Queensland (DAFF) trials at Condamine and Emerald did not show a yield response under conditions of terminal drought stress in spring.

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 7: 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)	Maize yield (kg/ha)	
		Nil (P & Zn)	+ (P & Zn)
21	14	2865	4937
11	26	3625	3632
6	44	5162	4704

Source: J Thompson (1984).

Table 8: Effect of fallow length and fertiliser on chickpea growth

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)	1.0	1.2	0.4	1.9
Short (6 months after wheat)	3.1	2.8	2.7	3.3

Results in Table 9 from a Queensland Wheat Research Institute trial at Macalister show that chickpea growth on short-fallow land (6 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. [17]

More information

www.grdc.com.au/GRDC-FS-PhosphorusManagement

http://www.icanrural.com.au/newsletters/NL52.pdf

D Lester, M Bell, R Graham, D Sands, G Brooke (2016), Phosphorus and potassium nutrition

5.5.4 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 (see symptoms of K deficiency in Figure 6).

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

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. [18]

Figure 7: Potassium deficiency symptoms in chickpea leaves. (Source: CSIRO)

More information

http://anz.ipni.net/article/ANZ-3193

5.5.5 Sulfur

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, when double-cropping chickpeas after high-yielding sorghum or cotton crops (see Figure 7 for symptoms of S deficiency).

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 (e.g. after sorghum or cotton due to sulfur removal rates). [19]

More information

G Blair, W Matamwa, I Yunusa, M Faint (2015), Adding sulfur to finished fertilisers: inside or outside?

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

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

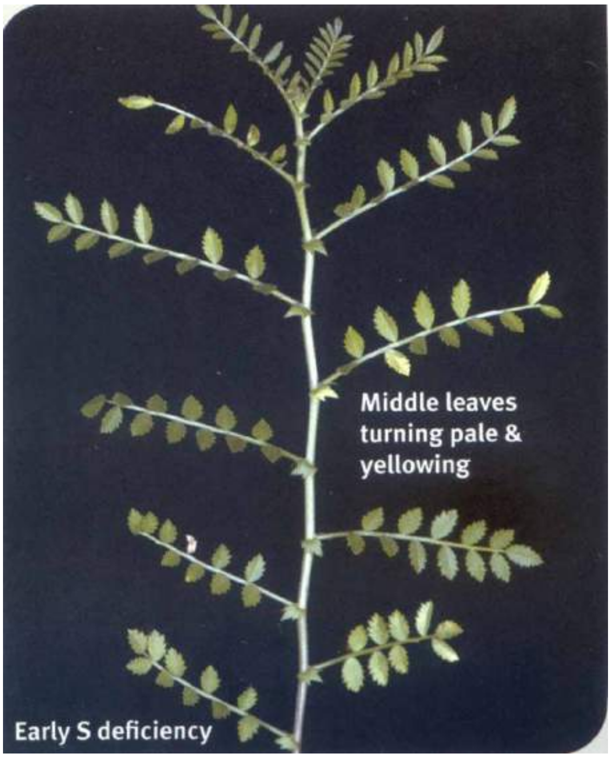

Figure 8: Early sulfur deficiency symptoms in chickpea leaves. (Source: CSIRO)

5.5.6 Zinc

Chickpeas are considered to have a relatively high demand for Zn, but have evolved highly efficient mechanisms for extracting Zn from the soil (similar to the mechanisms described for P).

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

AMF are extremely 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) (see deficiency symptoms in Figure 8).

More information

A Verrell, L Jenkins (2016). Nutrition in chickpea 2015 (northern NSW pulse agronomy project)

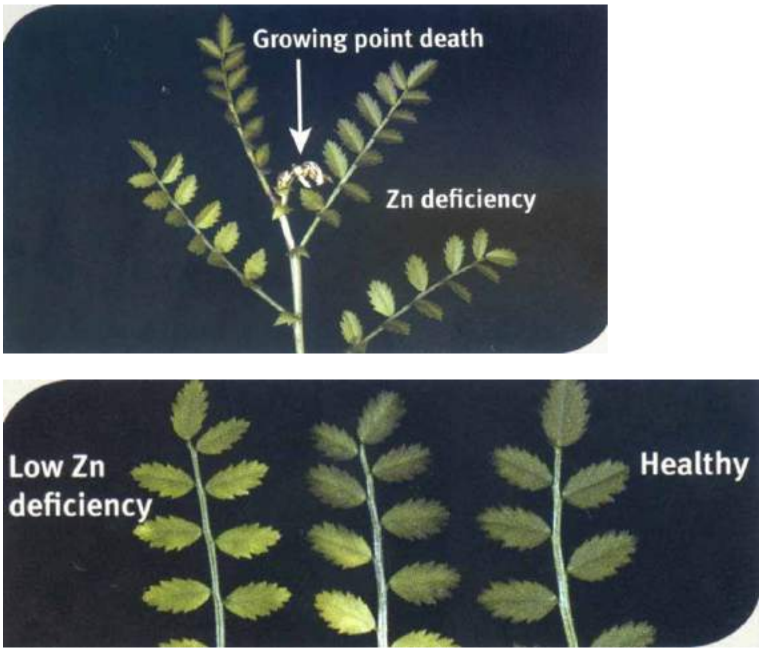

Figure 9: Comparisons of healthy and zinc-deficient chickpea leaves. (Source: CSIRO)

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.

Seed treatments

Zinc seed treatments may be a cost-effective option where soil P levels are adequate but Zn levels are likely to be deficient:

- Broadacre Zinc (Agrichem): contains 650 g/L of Zn and is applied as 4 L product /t seed. Pre-mix with 1 L water prior to application. To minimise damage to the rhizobia, the Broadacre Zinc treatment needs to be applied first and then allowed to dry before applying the inoculum. Broadacre Zinc is compatible with Thiraflo or P-Pickel T® and can be mixed with either product to treat chickpea seed in the one operation.
- Teprosyn Zn (Phosyn): contains 600 g/L of Zn and is applied as 4 L product/t seed. Pre-mix with 2–3 L water to assist coverage. Apply inoculum first and allow to dry before applying the Teprosyn.

Fertilisers applied at sowing

A range of phosphate-based fertilisers either contain, or can be blended with, a Zn additive.

Foliar zinc sprays

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. [20]

5.5.7 Iron

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.

Symptoms include a general yellowing of young leaves, which can develop in severe cases to distortion, necrosis and shedding of terminal leaflets (pinnae) (Figure 9).

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.

Figure 10: Iron deficiency in chickpeas. (Source: CSIRO)

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

Cultivars exhibit marked differences in sensitivity to iron chlorosis, and 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. [21]

5.6 References and further reading

IP Abrol, KS Dargan, DR Bhumbla (1973) Reclaiming alkali soils. Bulletin 2, Central Soil Salinity Research Institute, Karnal, India.

JB Dickie, RH Ellis, HL Kraak, K Ryder, PB Tompsett (1990) Temperature and seed storage longevity. Annals of Botany 65(2), 197–204.

RH Ellis, K Osei-Bonsu, EH Roberts (1982) The influence of genotype, temperature and moisture on seed longevity in chickpea, cowpea and soya bean. Annals of Botany 50(1), 69–82.

N Ferguson, G Schwenke, I Daniells, B Manning (2006) Adapting crop species on grey Vertosols in North West NSW to compensate for high chloride levels in the subsoil. In Proceedings 13th Australian Agronomy Conference, www.regional.org.au/asa/2006/concurrent/systems/4635_ferguson.htm

GRDC (2008) 'Grain legume handbook for the pulse industry'. Grain Legume Hand Book Committee, supported by the Grains Research and Development Corporation (GRDC)

L Jenkins, K Moore, G Cumming (2011) Chickpea: sourcing high quality seed. Northern Pulse Bulletin. Pulse Australia, www.pulseaus.com.au/pdf/Chickpea%20Sourcing%20High%20Quality%20Seed.pdf

EV Maas, EJ Hoffman (1977) Crop salt tolerance, current assessment. Journal of Irrigation and Drainage ASCE 103, 115–134.

Queensland Natural Resources and Water (2007) Constraints to cropping soils in the northern grains region—A decision tree. Queensland Natural Resources and Water Bulletin, http://www.grdc.com.au/uploads/documents/dnr00004.pdf

FW Smith, WHJ Pieters (1983) Foliar symptoms of nutrient disorders in chickpea (*Cicer arietinum*). Technical Paper No. 23. CSIRO Tropical Crops and Pastures.

E Drew, D Herridge *et al.* (2012) 'Inoculating legumes: A Practical guide.' GRDC, www.grdc.com.au/~/media/E4709CA6DB6F4A408952E42651D340E3.pdf

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

SECTION 6

Weed control

Videos

http://www.grdc.com.au/Media-Centre/Media-News/North/2013/05/Harvest-weed-seed-control-key-to-overcoming-resistance

More information

http://www.grdc.com.au/uploads/documents/GRDC_HerbicideCard.pdf

http://www.dpi.nsw.gov.au/agriculture/pests-weeds/weeds/publications/nhrr

http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0006/155148/herbicide-resistance-brochure.pdf

NSW DPI, Weed control in winter crops 2015

Pulse Australia (2016), Chickpea production: northern region

Weeds are estimated to cost Australian agriculture AU$2.5–4.5 billion per annum. For winter cropping systems alone, the cost is $1.3 billion. Consequently, any practice that can reduce the weed burden is likely to generate substantial economic benefits to growers and the grains industry. See more at www.grdc.com.au/weedlinks. [1]

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. [2]

The Grains Research and Development Corporation (GRDC) supports integrated weed management. Download the Integrated Weed Manual.

Weed control is important, because weeds can:

- 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
- prevent some crops being grown where in-crop herbicide options are limited, i.e. broadleaf crops
- be toxic to stock
- carry disease
- host insects

6.1 Planning your weed control strategy

More information

http://www.rbgsyd.nsw.gov.au/plant_info/identifying_plants/plant_identification_service

1. Know your weed species. Ask your local adviser or service provider, or use the Sydney Botanic Gardens plant identification service, which is free in most cases (see link).
2. Conduct in-crop weed audits prior to harvest to know which weeds will be problematic the following year.
3. Ensure that seed is kept from a clean paddock (Figure 1).
4. Have a crop-rotation plan that considers not just crop type being grown but also the weed-control options this crop system may offer (e.g. grass control with triazine-tolerant (TT) canola).

1 GRDC (2005) Weedlinks. Integrated weed management. GRDC, www.grdc.com.au/weedlinks

2 DAFF (2012) Wheat—planting information. Department of Agriculture, Fisheries and Forestry, Queensland, http://www.daf.qld.gov.au/plants/field-crops-and-pastures/broadacre-field-crops/wheat/planting-information

Figure 1: Ensure that chickpea seed is kept from a clean paddock. (Photo: G. Cumming, Pulse Australia)

6.2 Herbicide resistance

Herbicide resistance is an increasing threat across Australia's northern grain region for growers and agronomists. Already, 14 weeds have been confirmed as herbicide-resistant in various parts of this region, and more have been identified at risk of developing resistance, particularly to glyphosate.

In northern New South Wales (NSW), 14 weeds are confirmed resistant to herbicides of Group A, B, C, I, M or Z (Table 1). Barnyard grass, liverseed grass, common sowthistle and wild oat have confirmed cases of resistance to Group M (glyphosate) herbicides (Table 2). Glyphosate-resistant annual ryegrass has been identified within about 80 farms in the Liverpool Plains area of northern NSW (Figure 2). [3]

Table 1: List of confirmed resistant weeds in northern NSW (as at November 2016)

Weed	Herbicide group and product/chemical (examples only)	Areas with resistance in NSW	Future risk	Detrimental impact
Wild oats	A. Topik® and Wildcat® B. Atlantis® Z. Mataven®	Spread across the main wheat-growing areas. More common in western cropping areas	Areas growing predominantly winter crops	High
Paradoxa grass	A. Wildcat®	North and west of Moree	Areas growing predominantly winter crops	High
Awnless barnyard grass	C. Triazines M. Glyphosate	Mainly between Goondiwindi and Narrabri	No-till or minimum tilled farms with summer fallows	High Very high
Charlock, black bindweed, common sowthistle, Indian hedge mustard, turnip weed	B. Glean®, Ally®	Spread across the main wheat growing areas	Areas growing predominantly winter crops	Moderate
Annual ryegrass	M. Glyphosate B. Glean® A. Verdict®	Group M widespread in Liverpool Plains. Group A and B resistance in central west NSW	Areas with predominantly summer fallows. Winter cropping areas	High High
Fleabane	M. Glyphosate	Spread uniformly across the region	Cotton crops and no-till or minimum tilled systems	Moderate

[3] A Storrie, T Cook, P Moylan, A Maguire, S Walker, M Widderick. Managing herbicide resistance in northern NSW, NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0006/155148/herbicide-resistance-brochure.pdf

Weed	Herbicide group and product/chemical (examples only)	Areas with resistance in NSW	Future risk	Detrimental impact
Wild radish	I. 2,4-D amine	Central west NSW	Continuous winter cereal cropping	High
Windmill grass	M. Glyphosate	Central west NSW	Continuous winter cropping and summer fallows	High
Liverseed grass	M. Glyphosate	A few isolated cases	No-till or minimum tilled systems	Moderate
Sowthistle	M. Glyphosate	Liverpool Plains	Winter cereal dominated areas with minimum tillage	High
Feather-top Rhodes grass	M. Glyphosate	Widespread, more common in the north	No-till or minimum tilled systems, sorghum and cotton crops	High

Table 2: List of potential new resistant weeds in northern NSW (as at November 2016)

Weed	Herbicide group and product/chemical (examples only)	Future risk	Detrimental impact
Barnyard, liverseed and windmill grasses	A. Verdict® L. Paraquat	No-till and minimum tilled systems	Very high Very high
Common sowthistle	I. 2,4-D amine	Winter cereals	High
Paradoxa grass	B. Glean®, Atlantis®	Western wheat growing areas	High
Other brassica weeds including wild radish	B. Glean®,, Ally®	Areas growing predominantly winter crops	Moderate
Annual ryegrass	L. Paraquat	Areas with predominantly summer fallows	Very high
Wireweed, black bindweed, melons and cape weed	I. 2,4-D amine, Lontrel®, Starane®	Areas growing predominantly winter crops	High
Fleabane	I. 2,4-D amine	Cotton crops and no-till or minimum tilled systems	Very high
Other fallow grass weeds	M. Glyphosate	No-till or minimum tilled systems	High

More information

http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0006/155148/herbicide-resistance-brochure.pdf

http://www.grdc.com.au/uploads/documents/GRDC_NorthernWeeds.pdf

M Street, B O'Brien (2016), Report on the 2014 GOA herbicide resistance survey

Figure 2: Glyphosate resistant annual ryegrass on the Liverpool Plains, NSW. (Photo: D. Freebairn)

Testing services

For testing of suspected resistant samples, contact:

Charles Sturt University Herbicide Resistance Testing
School of Agricultural and Wine Sciences
Charles Sturt University
Locked Bag 588
Wagga Wagga, NSW 2678
02 6933 4001
https://www.csu.edu.au/__data/assets/pdf_file/0009/1688904/2015-report.pdf

Plant Science Consulting
22 Linley Ave
Prospect, SA 5082
0400 664 460
info@plantscienceconsulting.com.au
www.plantscienceconsulting.com

6.2.1 Be a WeedSmart farmer

Figure 3: WeedSmart logo.

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 (Figure 3).

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 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

Videos

Richard Daniel, NGA, discusses herbicide resistance tactics: http://www.weedsmart.org.au/media-releases/rethinking-weed-control/

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. [4]

6.2.2 Ten ways to weed out herbicide resistance

1. Act now to stop weeds from setting seed:

- Destroy or capture weed seeds.
- Understand the biology of the weeds present.
- Remember that every successful WeedSmart practice can reduce the weed seedbank over time.
- Be strategic and committed—herbicide resistance management is not a 1-year decision.
- Research and plan your WeedSmart strategy.
- You may have to sacrifice yield in the short term to manage resistance—be proactive.
- Find out what other growers are doing, and visit www.weedsmart.org.au.

2. Capture weed seeds at harvest. Options to consider are:

- Tow a chaff cart behind the header.
- Check out the new Harrington Seed Destructor.
- Create and burn narrow windrows.
- Produce hay where suitable.
- Funnel seed onto tramlines in controlled traffic farming (CTF) systems.
- Use crop-topping where suitable (southern and western grains region).
- Use a green or brown manure crop to achieve 100% weed control and build soil nitrogen levels.

3. Rotate crops and herbicide modes of action:

- Look for opportunities within crop rotations for weed control.
- Understand that repeated application of effective herbicides with the same mode of action (MOA) is the single greatest risk factor for evolution of herbicide resistance.
- Protect the existing herbicide resource.
- Remember that the discovery of new, effective herbicides is rare.
- Acknowledge that there is no quick chemical fix on the horizon.
- Use break-crops where suitable.
- Growers in high-rainfall zones should plan carefully to reduce weed populations in the pasture phase prior to returning to cropping.

4. Test for resistance to establish a clear picture of paddock-by-paddock weed status:

- Sample weed seeds prior to harvest for resistance testing to determine effective herbicide options.
- Use the 'Quick Test' option to test emerged ryegrass plants after sowing to determine effective herbicide options before applying in-crop selective herbicides.
- Visit the WeedSmart website, www.weedsmart.org.au or www.ahri.uwa.edu.au for more information on herbicide-resistance survey results.
- Collaborate with researchers by collecting weeds for surveys during the double-knock program (northern region).

[4] WeedSmart, http://www.weedsmart.org.au

5. Aim for 100% weed control and monitor every spray event:

- Stop resistant weeds from returning into the farming system.
- Focus on management of survivors in fallows (northern grains region).
- Where herbicide failures occur, do not let the weeds seed. Consider cutting for hay or silage, fallowing or brown manuring the paddock.
- Patch-spray areas of resistant weeds only if appropriate.

6. Do not automatically reach for glyphosate:

- Use a diversified approach to weed management.
- Consider post-emergent herbicides where suitable.
- Consider strategic tillage.

7. Never cut the on-label herbicide rate and carefully manage spray drift and residues:

- Use best management practice in spray application. The GRDC has produced a series of Fact Sheets, available at www.grdc.com.au.
- Consider selective weed sprayers such as WeedSeeker or WeedIt.

8. Plant clean seed into clean paddocks with clean borders:

- It is easier to control weeds before the crop is planted.
- Plant weed-free crop seed to prevent the introduction of new weeds and the spread of resistant weeds.
- A recent Australian Herbicide Resistance Initiative survey showed that 73% of grower-saved crop seed was contaminated with weed seed.
- The density, diversity and fecundity of weeds are generally greatest along paddock borders and areas such as roadsides, channel banks and fence lines.

9. Use the double-knock technique:

- Double-knock technique is the use of any combination of weed control that involves two sequential strategies; the second application is designed to control survivors of the first method of control used.
- Access GRDC research results at www.grdc.com.au or www.nga.org.au.

10. Employ crop competitiveness to combat weeds:

- Consider narrow row spacing and increased seeding rates.
- Consider twin-row seeding points.
- Use barley, canola and varieties that tiller well.
- Use high-density pastures as a rotation option.
- Consider brown manure crops.
- Rethink bare fallows. [5]

More information

www.weedsmart.org.au

http://www.ahri.uwa.edu.au

http://www.grdc.com.au/BGC00001

6.3 Harvest weed-seed control

Controlling weed seeds at harvest is emerging as the key to managing the increasing levels of herbicide resistance that are putting Australia's no-till farming system at risk.

For information on harvest weed-seed control and its application for the northern grains region, see *GrowNotes (Chickpeas) Section 12: Harvest*.

More information

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2012/04/Harvest-weed-seed-control

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2012/04/What-percent-of-northern-weed-seed-might-it-be-possible-to-capture-and-remove-at-harvest-time-A-scoping-study

5 WeedSmart, http://www.weedsmart.org.au

6.4 Other non-chemical weed control

Crop rotation, especially with summer crops, can be an effective means of managing a spectrum of weeds that result from continuous wheat cropping. Barley is a more vigorous competitor of weeds than is wheat, and it may be a suitable option for weed suppression. Increased planting rates and narrow rows may also help where the weed load has not developed to a serious level. [6]

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. For the management of wild oats, the inclusion of a strategic summer crop such as sorghum means two winter fallows, with glyphosate an option for fallow weed control. Grazing and/or cultivation are alternative, non-chemical options.

Where continuous summer cropping has led to development of Group M resistant annual ryegrass, a winter crop could be included in the rotation and a Group A, B, C, D, J or K herbicide used instead, along with crop competition and potential harvest-management tactics.

For summer grasses, consider a broadleaf crop such as mungbean, because a Group A herbicide and crop competition can provide good control.

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.

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. [7]

Videos

http://www.youtube.com/playlist?list=PL2PndQdkNRHGRipNhkDYN2dJWAY1-oH9W

http://www.grdc.com.au/Media-Centre/Ground-Cover-Supplements/GCS104/Lift-sowing-to-outcompete-weeds

http://www.ahri.uwa.edu.au

http://www.agronomo.com.au/giving-a-rats/

More information

http://www.grdc.com.au/Media-Centre/Hot-Topics/Herbicide-Resistance

6.5 Herbicides explained

6.5.1 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 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.

More information

L van Zwieten et al (2016). Herbicide residues in soils – are they an issue? (Northern)

6.5.2 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,

[6] DAFF (2012) Wheat—planting information. Department of Agriculture, Fisheries and Forestry, Queensland, http://www.daf.qld.gov.au/plants/field-crops-and-pastures/broadacre-field-crops/wheat/planting-information

[7] GRDC (2012) Herbicide resistance. Cropping with herbicide resistance. GRDC Hot Topics, http://www.grdc.com.au/Media-Centre/Hot-Topics/Herbicide-Resistance

whereas pre-emergent refers to application of the herbicide to the soil before the weeds have emerged. [8]

6.6 Integrated weed management in chickpea

Good weed control management is vital to successful and profitable crop production. Yield losses caused by weeds can vary enormously, from almost negligible to a complete loss.

Weeds lower crop yields by competing for soil moisture, nutrients, space and light and can carry diseases that attack crops. This competition reduces grain yield and quality, and can impede harvesting. Some weeds can restrict cropping options, as herbicides for control are sometimes limited. Investigate which weed species are likely to germinate in a paddock before sowing crops and determine the availability of suitable herbicide options.

Weed control is a numbers game. Growers should aim to reduce weed numbers and keep them low with an ongoing program. A weed-management program should make the most of rotations and hence opportunities to use selective herbicides from a different herbicide group in each crop in the rotation to reduce the weed problem in the following crop. Care should be taken in planning a cropping rotation to avoid herbicide resistance, or growing a crop that may become a 'weed' or lead to uncontrolled weeds that cannot be controlled with selective herbicides in the subsequent crop.

An integrated weed management system combining all of the available methods is the key to successful control of weeds. [9]

Videos

Videos: IWM—Integrated weed management videos: http://www.youtube.com/playlist?list=PL2PndQdkNRHGRipNhkDYN2dJWAY1-oH9W

Crop rotation

A well-managed rotation in each paddock, which alternates pastures and broadleaf and cereal crops, is a very useful technique for controlling weeds. For example, chemical control of grass weeds is easier and cheaper in broadleaf crops, whereas control of broadleaf weeds is much easier in cereal crops. Good crop rotation management can substantially reduce the cost of controlling weeds with chemicals.

Pulses grown in rotation with cereal crops offer opportunities to control grassy weeds with selective herbicides that cannot be used in the cereal phases. This is possible only if the grower can still effectively use Group A chemistry. An effective kill of grassy weeds in pulse crops will reduce carry-over of root disease and provide a 'break crop' benefit in following cereal crops. Grass-control herbicides can control most grassy weeds in pulses. Volunteer cereals can also be controlled with some of these herbicides.

Good agronomic practice

Practices such as using weed-free seed (preferably registered or certified) and sowing on time with optimal plant populations and adequate nutrition all contribute to good weed-control management. Some crops and varieties are more competitive than others against weeds. All weeds growing in a paddock should be controlled before the crop emerges. Large weeds that have not been controlled prior to, or by, the sowing operation prove most difficult and often impossible to remedy with in-crop herbicides.

Timely cultivation

Timely cultivation is a valuable method for killing weeds and preparing seedbeds. Some growers use combinations of mechanical and chemical weed control to manage their fallows or stubbles. Increasing numbers of growers are using knockdown herbicides instead of cultivation for fallow commencement, as well as for pre-planting weed control in autumn.

8 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, http://www.grdc.com.au/~/media/A4C48127FF8A4B0CA7DFD67547A5B716.pdf

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

These practices are providing clear benefits to soil structure, as well as more timely and effective weed control.

In-crop weed control

A wide range of pre-emergent and early post-emergent herbicides is available for in-crop weed control. Weeds should be removed from crops as early as possible, certainly no later than 6 weeks after sowing, if yield losses are to be minimised. Yield responses will depend on weed species, weed and crop density, and seasonal conditions. The growth stage of the weed and the crop are vital considerations when planning the use of post-emergent herbicides. Read herbicide labels carefully for relevant details and information on the best conditions for spraying.

http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0007/431269/Fleabane-management-in-crop-rotations.pdf

http://www.dpi.nsw.gov.au/agriculture/broadacre/guides/ngrt-results

Herbicide resistance

Herbicide resistance is a problem that is becoming more widespread, and growers should be alert. It is one of the biggest agronomic threats to the sustainability of our cropping systems. However, the problem can be managed with good crop rotations, by rotating herbicide groups and by combining chemical and non-chemical methods of weed control (Table 3).

Options for control of broadleaf weeds with selective herbicides in chickpeas are generally limited compared with the treatments available for use in cereal crops.

Table 3: Weed control options for integrated weed management (IWM)

	Herbicidal	Non-herbicidal
Crop phase	Crop-topping in pulse/legume crops Knockdown herbicides, e.g. double-knock strategy before sowing Selective herbicides before and/or after sowing but ensuring that escapes do not set seed Utilising moderate resistance risk herbicides Delayed sowing (as late as spring in some cases) with weeds controlled in the interim	Rotating crops Rotating varieties Growing a dense and competitive crop Cultivation Green/brown manure crops Delayed sowing Cutting crops for hay/silage Burning stubbles/windrows. Chickpeas offer a very good opportunity to utilise this tactic. The lower biomass of stubble makes fires easier to manage Collecting weed seeds at harvest and remove/burn Destroying weed seeds harvested (Use of Harrington Seed destructor)
Pasture phase	Spray topping Winter cleaning Selective herbicides but ensuring that escapes do not set seed	Good pasture competition Hay making or silage Cultivated fallow Grazing

Keep yourself informed and be pro-active in the prevention of, or fight-back against, resistance. [10]

More information

For further information on resistance management strategies refer to Integrated Weed Management Manual on the following websites:

www.croplifeaustralia.org.au

www.glyphosateresistance.org.au

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

6.7 Weed issues specific to chickpeas

Chickpeas are poor competitors with weeds because of slow germination and early growth. There are limited options for pre-emergent and post-emergent weed control. Chickpeas should always be planted into planned paddocks that have low weed populations. Please consult your agronomist for further information.

6.8 Broadleaf weed control

Paddock selection and effective application of pre-emergent broadleaf herbicide is critical in chickpea. Paddocks with a severe broadleaf weed problem should be avoided because broadleaf weed control is very limited in chickpeas.

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 (e.g. double-cropped from sorghum or cotton, or country with a sequence of clean winter fallows).

Post-emergent herbicides

Only one herbicide is currently registered for post-emergent use in chickpeas, Broadstrike® (active ingredient flumetsulam), and caution must be taken with its use. Broadstrike® gives good control of cruciferous species (turnip, etc.) but has no activity on many thistle species such as sow thistle and prickly lettuce. Broadstrike® can result in significant crop damage and can delay harvest, as is clearly stated on the label. Broadstrike® should mainly be used in salvage situations and even then should only be applied under conditions of good soil moisture and on very small, actively growing weeds, as per the label.

More information

www.apvma.gov.au/pubcris

Pre-emergent herbicides

In the absence of safe post-emergent broadleaf herbicides, control is limited to a few pre-emergent herbicides such as Balance® (active ingredient isoxaflutole), simazine and Terbyne® (active ingredient terbuthylazine). Most of these chemicals are very dependent on rainfall for activation; hence, results are often limited under dry conditions.

Pre-emergent herbicides for use in chickpeas are generally registered for either incorporation by sowing or use as a post-sowing pre-emergent (PSPE). Please read the labels carefully to minimise the chance of crop damage.

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. 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.

6.9 Post-emergent grass weed control

Control of post-emergent grass weeds is often inconsistent, with variable levels of control achieved. This particularly applies to many broadacre situations where marginal rates of Group A herbicides are being used.

More reliable and cost-effective control can be achieved through the adoption of a management package that addresses all of the following key issues:

- Correctly identify the weed.
- Match the product used to the weeds present.

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

- Weeds should be controlled when small, preferably at the 2–5 leaf stage.
- Larger weeds will require higher rates of Group A herbicides.
- Spray when weeds are actively growing and free from temperature, water, and nutritional stress.
- Weeds enter into moisture stress quickly, especially if secondary roots have not established.
- The leaves can also become water repellent under dry, dusty conditions.
- Seedling grasses stress very quickly and usually there is a narrow window of ideal conditions for applying Group A herbicides.
- Application techniques and boom sprayer setup are critical in achieving coverage of seedling grasses.
- Nozzle selection should be made to achieve a medium spray quality.
- Operating pressures should be >3 bar.
- Water volumes should be >60 L/ha.
- Use the preferred adjuvant listed on the product label.
- Know your resistance status. [12]

6.9.1 Mode of action

All of the grass herbicides are systemic and rely on absorption through the leaves and translocation to the growing points (meristematic tissue) of the plant. Treated grasses usually stop growing within 1–2 days of spraying.

Visible symptoms first appear 7–10 days after treatment, usually as a yellowing of the youngest leaves and a browning of the growing points at the base of the youngest leaves. Unfurled leaves are easily pulled out, revealing brown rotting buds at the leaf base.

The young leaves turn pale and chlorotic and then brown off. The older leaves eventually collapse, with complete plant death occurring 4–6 weeks after spraying. Some weed species may also exhibit reddening of lower leaves and leaf sheaths.

More information

http://www.dpi.nsw.gov.au/agriculture/broadacre/guides/weed-control-winter-crops

6.9.2 Managing wild oats in chickpeas

Chickpea rotations provide an opportunity to control wild oats, which is otherwise a costly weed in a wheat-based system. [13] 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 northern 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.

Chickpeas are the major northern winter rotation crop; therefore, any threat to chickpea production area could have a major impact on our regional farming system.

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

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

[13] DAFF (2012) Chickpea—weed management. Department of Agriculture, Fisheries and Forestry, Queensland, http://www.daf.qld.gov.au/plants/field-crops-and-pastures/broadacre-field-crops/chickpeas/weed-management

including knockdowns) as well as improved crop competition to reduce seed-set of wild oats.

Winter fallow leading into, or out of, sorghum and other summer crops

Winter fallow enables the use of low-risk knockdown herbicides such as glyphosate and paraquat to control the multiple wild oat germinations that will emerge through winter and spring. Atrazine can also be applied in spring before sorghum planting to assist in the control of late-emerging wild oats.

Two summer crops in succession will reduce the wild oat seedbank, particularly in a no-till system. This is primarily because weed seeds are removed from the seedbank faster if they are left on the soil surface. These seeds are exposed to fluctuations in temperature and moisture, which increases the rate of seed decay. Figure 4 shows that the seedbank of 'fop'-resistant wild oats declined to very low numbers after 2 years of total seed-set control in a no-till fallow. Starting with a higher initial seedbank would increase the time taken to reduce the seedbank to manageable levels for chickpeas. [14]

Wild oat seeds are relatively short-lived, with a half-life of about 6 months. This effectively means that if no new seed is allowed to develop, half of the seed reserves are depleted in 6 months, 75% in 12 months, and >92% in 2 years.

Maximum reduction of oat reserves will be achieved if seed-set is prevented over the course of two winter seasons, for example, by combining oat control in chickpeas with the inclusion of a sorghum crop in the rotation and a clean winter fallow.

More information

http://www.nga.org.au/results-and-publications/download/45/australian-grain-articles/weeds-1/wild-oats-in-chickpeas-tip-of-the-iceberg-september-2007.pdf

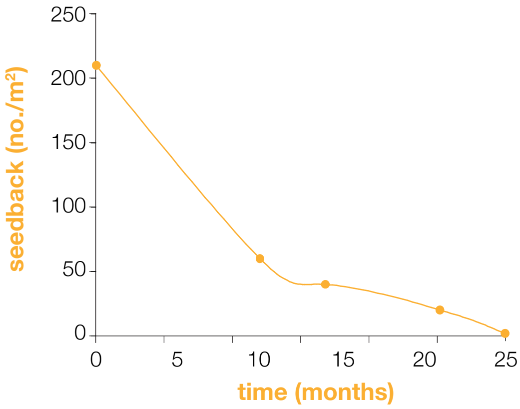

Figure 4: Decline in wild oat seedbank with total seed-set control, North Star, New South Wales.

6.9.3 Avoidance of stress conditions

All grass herbicides labels emphasise the importance of spraying only when the weeds are actively growing under mild, favourable conditions. 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®

[14] A 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-in-chickpeas-tip-of-the-iceberg-september-2007.pdf

- excessively heavy dews resulting in poor spray retentions on grass leaves

Ensure that grass weeds have fully recovered before applying grass herbicides.

Research from overseas has verified that translocation rates of fluazifop are 2–3 times higher in oats grown under high nitrogen status than in low-fertility situations (Table 4). [15]

Table 4: Impact of low nitrogen fertility on translocation of fluazifop

	Uptake	Translocation	Translocated to youngest leaf (dpm^A/mg)
	(% applied dose)		
Low nitrogen status	69%	9%	8
High nitrogen status	63%	26%	24

Source: Dickson et al. 1990.
^A Disintegrations per minute.

6.9.4 Grass herbicide damage in chickpeas

Group A herbicides can occasionally cause leaf spotting in chickpeas (Figure 5). This is usually associated with either frost or high temperatures occurring soon after spray application. [16]

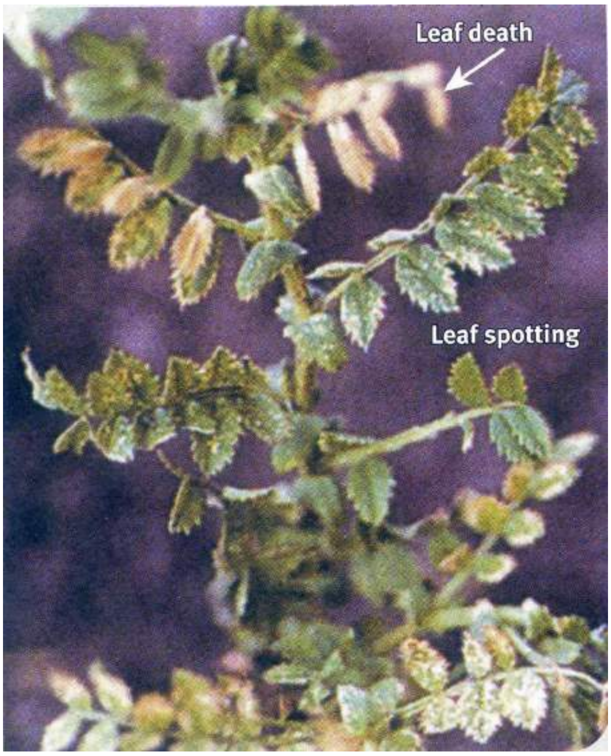

Figure 5: Group A grass selective herbicide injury. (Photo: T. Bretag)

6.9.5 Sulfonylurea residues in boom sprays

Traces of sulfonylurea (SU) herbicides in boom sprays have the potential to cause significant damage to chickpea crops (Figure 6). The risk of residue damage is greater in the presence of grass-selective herbicides.

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

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

Decontaminate the boom if you have previously used an SU herbicide. See product labels for specific product recommendations on decontamination.

Figure 6: Damage to field peas from failing to decontaminate the spray tank after use of Eclipse®.

6.10 Weed control requires a planned approach

Chickpeas are late-maturing compared with other pulses; hence, crop-topping to prevent ryegrass and other weed seed-set is not possible, even in the earliest maturing varieties (e.g. Genesis 079). Chickpeas are relatively slow to emerge, with slow early growth during the colder, winter months. Consequently, they are poor competitors with weeds. Even moderate weed infestations can cause large yield losses and harvest problems.

Trials in northern New South Wales and central Queensland have shown that populations of 5–10 turnip or 10 wild oat plants per m2 can cut yields by as much as 40–60% (D White 2000; J Whish 1998). Because of the slow growth and open canopy of chickpeas, narrow or wide row-spacing (30 *v.* 70 cm) made little difference to the chickpea plant's ability to compete with weeds.

Broadleaf weed control options can be very limited in chickpeas, and this is a reason producers commonly give for not growing chickpeas.

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.

Selection of paddocks that are relatively free or carry only a low burden of grass and broadleaf weeds is important.

Broadleaf weeds must be heavily targeted in the preceding crop and/or fallow. Always assess the broadleaf weed risk prior to planting.

This should be based on:

- grower's experience
- the previous crop and herbicides used
- an assessment of winter weeds germinating in the fallow prior to planting

Paddocks with a severe broadleaf or grass weed problem should be avoided. [17]

6.10.1 Knockdown herbicides

The most important part of a weed-control strategy is to 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

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

germinating weeds before the crop has time to emerge. Chickpea crops may take up to 21 days to emerge under cool, drying soil conditions, but under favourable warm, moist soil conditions, they may emerge after 7 days. Growers considering this option should sow deeper (10–15 cm) and carefully check their paddocks for the emergence of the chickpeas immediately before spraying. Done correctly, this can be an effective weed-control option. [18]

6.10.2 Pre-emergent herbicides

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. Best weed control is achieved from PSPE application because rainfall gives the best incorporation. Mechanical incorporation pre-sowing is less uniform, and so weed control may be less effective. 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.

More information

[J Cameron, M Congreve (2016). Recropping issues with pre emergent herbicides]

[C Preston, S Kleemann, G Gill (2016). Coupling pre emergent herbicides and crop competition for big reductions in weed escapes]

[R Daniel (2016). Pre emergent herbicides: part of the package for FTR management?]

Weed control

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 dependent on rainfall soon after application and, consequently, often result in inconsistent or partial weed control under drier conditions.

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.

Crop safety

The safety of chickpea crops will depend in part on chemical tolerance of the crop and variety, in part on ensuring that the seed is below the treated soil, and in part on ensuring that there is no wash of herbicide into the seeding furrow.

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.

The pH of a soil can 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.

The movement of herbicides down the soil profile after rain can affect crop safety. Movement is greater on sandy soils (and those with less organic matter), and so the rate must be lower than on heavier soils (loams, silt plus clay 40–60%).

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

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

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. [19]

6.10.3 Pre-sowing (incorporation by sowing) herbicides

Herbicides that may be used in some situations with chickpeas include trifluralin (i.e. TriflurX®), pendimethalin (i.e. Stomp®), triallate (i.e. Avadex®), cyanazine (i.e. Bladex®), simazine and some diuron brands (e.g. Diurex®); these are registered for use on chickpea. Most require mechanical incorporation by sowing, and they are often used in mixtures.

More information

www.apvma.gov.au/pubcris

Registrations differ broadly across states, formulations and soil types. This is particularly true for weeds falling into the suppression ranges rather than those registered for outright control. Please consult your agronomist and read the labels of the products you are considering using. These herbicide labels also have information on the timing of incorporation, advice on how to avoid damage, as well as plant-back restrictions.

Most products work best if thoroughly mixed 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. Some herbicide incorporation occurs when sowing is done with knife-points, provided sowing speed is adequate to throw soil 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.

With the continued development of Group A, Group B and Group D resistant populations of annual ryegrass and wild oats, growers are again using these 'older' products as part of their resistance strategy because of the opportunity that they provide to rotate chemical groups.

6.10.4 Post-sowing pre-emergent (PSPE) herbicides

Herbicides that may be suitable for PSPE applications to chickpeas include simazine, Balance®, Terbyne®, prometryn and diuron. As with pre-sowing herbicides, registrations differ broadly across states, formulations and soil types, and the same advice applies regarding consulting your agronomist and reading product labels.

6.10.5 Post-emergent herbicides

Flumetsulam (e.g. Broadstrike®) can result in significant crop damage in our environment, particularly if dry conditions are experienced after application. As stated on the product label, Broadstrike® usually causes some transient crop yellowing and can cause reddish discoloration and height suppression. Flowering may be delayed (Figure 7), resulting in yield suppression.

Broadstrike® is used mainly in salvage situations (as a last resort), and even then should be applied only under good growing conditions. Figure 8 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.

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

Figure 7: 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)

Figure 8: 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)

6.11 Other weed control strategies

6.11.1 Directed sprays in-crop

With the shift to row-cropping chickpeas on wide rows, there is increasing adoption 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.

6.11.2 Shielded sprayers

These are becoming increasingly more common in or around cotton-growing areas, as they provide very cheap control of grass and broadleaf weeds with glyphosate.

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) [20]

6.12 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 2 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.

6.13 Herbicide performance

Characteristics that determine herbicide performance and activity are:

- herbicide uptake—how and where the chemical is taken up by the plant
- herbicide solubility—how readily it dissolves or leaches in soil water
- herbicide adsorption—how much is lost by binding to the soil
- herbicide persistence—how long it lasts on the soil, affected by:
- volatility, loss to the atmosphere
- leaching potential, i.e. how much is lost below the root-zone
- decomposition by light

Understanding these factors will assist in ensuring effective use of herbicide. For best performance, some pre-emergence herbicides (i.e. trifluralin) should be incorporated into the top 0–7.5 cm of soil. They must enter the germinating weed seedling in order to kill it. These herbicides can be mixed in by cultivation, rainfall or sprinkler irrigation, depending on the chemical.

Poor herbicide efficacy can occur under dry conditions at application. Some soil-active herbicides (e.g. Balance® or simazine) can damage chickpeas where wetter conditions favour greater activity and leaching. [21]

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

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

6.14 Herbicide damage in pulse crops

The risk of crop damage from a herbicide should be balanced against the potential yield loss from weed competition. In heavy weed infestations, some herbicide crop damage can be tolerated as this is offset by the yield loss avoided by removing competing weeds.

If herbicide is applied to dry soils, the risk of crop damage is increased greatly when rains do occur, particularly if the soil is left ridged so that 'herbicide wash' into the seed row occurs. Incorporation by seeding may be more appropriate in dry conditions, or a split application to minimise risk. The PSPE herbicides should be applied to moist soil regardless of sowing time.

Herbicides move more readily in soils with low organic matter, or more sand, silt or gravel. Herbicide movement is much less in soils with higher organic matter and clay contents. Damage from leaching is also greater where herbicides are applied to dry, cloddy soils than to soils that have been left level and are moist on top from recent rainfall.

Herbicides have different leaching potentials, as shown by the leaching index (Table 5). For example, metribuzin leaches at almost three times the rate of simazine and seven times the rate of diuron.

Table 5: Relative leaching of some soil-active herbicides (where 1 = the least leaching)

Chemical	Example of product	Leaching index
Pendimethalin	Stomp®	1
Trifluralin	Treflan®	1
Diuron	Diuron	2
Prometryn	Prometryn	3–4
Simazine	Simazine	5
Metolachlor	Dual Gold®	6
Atrazine	Atrazine	10
Metribuzin	Sencor®	14

The relative tolerance of the crop type and variety will also affect crop damage from these herbicides.

For more specific details on soil-active herbicides and on the risk of crop damage in your cropping situation, seek advice from an experienced agronomist.

6.14.1 Symptoms of crop damage caused by herbicides

Symptoms of crop injury from herbicides do not always mean grain yield loss will occur. Recognition of crop injury symptoms allows the cause of the injury to be identified and possibly prevented in future crops. The type of injury depends on how the herbicide works in the plant, the site and seasonal conditions.

Herbicide injury may be obvious (e.g. scorched leaves) or it may be more subtle (e.g. poor establishment or delayed maturity) (Figures 9–14). Herbicide crop-injury symptoms can easily be confused with symptoms produced by other causes, such as frost (Figures 15 and 16), disease or nutrition.

Care should be taken when using crop oils and penetrants with herbicides, as these can increase the uptake of active chemicals and exceed crop tolerance. Always follow the herbicide label.

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.

Herbicide efficacy and crop safety of the new crop can suffer if the soil is dry at application time.

More information

For descriptions and pictures of herbicide injury refer to 'Field crop herbicide injury: The ute guide' and 'Chickpea disorders: The ute guide'. Both are available from GRDC Ground Cover direct: http://www.grdc.com.au/uploads/documents/GRDC-GroundcoverDirectCatalogue201105-101.pdf

Taking some general precautions can help to reduce the likelihood of crop damage with residual herbicide use:

- Do not apply 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/seedling.
- Avoid leaving an exposed, open slot over the seed with disc-openers and avoid a cloddy, rough tilth with tined-openers. [22]

Figure 9: Simazine post-sowing pre-emergent (PSPE) injury may be confused with frost injury. (Photo: G. Cumming, Pulse Australia)

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

Figure 10: Balance PSPE injury. (Photo: T. Knights, NSW DPI)

Figure 11: 2,4-D spray drift causing stem twisting. (Photo: G. Cumming, Pulse Australia)

Figure 12: Glyphosate spray drift, causing narrowing (spear) of new leaflets. (Photo: G. Cumming, Pulse Australia)

Figure 13: Trifluralin injury causing stunted growth and development of multiple growing points. (Photo: A. Mayfield Consulting)

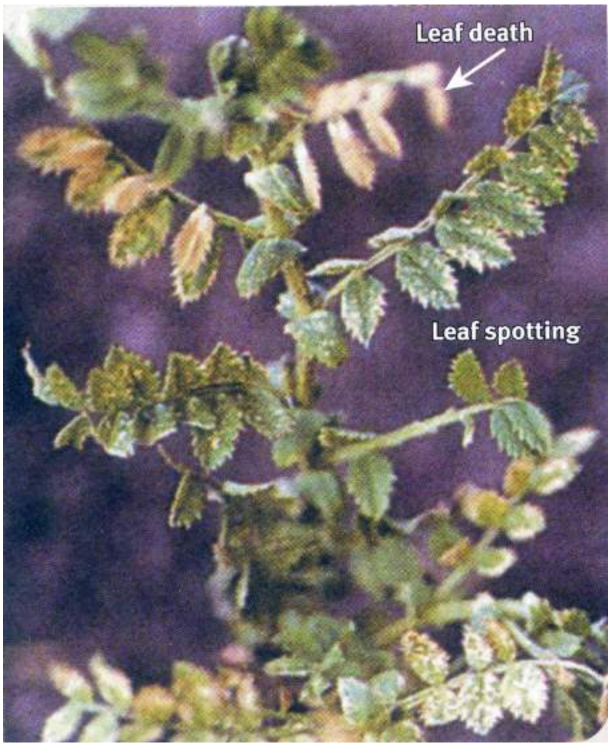

Figure 14: Group A grass-selective herbicide injury. (Photo: T. Bretag)

Figure 15: Frost damage. (Photo: G. Cumming, Pulse Australia)

Figure 16: Frost damage. Note the water soaking. (Photo: G. Cumming, Pulse Australia)

6.14.2 Contamination of spray equipment

The importance of cleaning and decontaminating spray equipment for the application of herbicides cannot be over-emphasised. Traces of SU 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. [23]

6.14.3 Spray drift

When applying pesticides the aim is to maximise the amount reaching the target and minimise the amount reaching off-target areas. This results in:

- maximum pesticide effectiveness
- reduced damage and/or contamination of off-target crops and areas

In areas where various agricultural enterprises co-exist, conflicts can arise, particularly from the use of pesticides.

Pulse crops can be severely damaged by some hormone herbicide sprays, such as 2,4-D ester, drifting into the crop. This can happen when those 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 hormone 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.

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

All pesticides are capable of drift. People have a moral and legal responsibility to prevent pesticides from drifting and contaminating or damaging neighbours' crops and sensitive areas (Figure 17). [24]

Figure 17: Glyphosate spray drift from the road verge on the left. Note the barrier effect of the tall weeds on the fence line. (Photo: G.D Bardell, Nufarm)

More information

http://www.weedsmart.org.au/media/watch-out-for-residuals/

6.15 Getting best results from herbicides

Successful results from herbicide application depend on numerous interacting factors. Many of the biological factors involved are not fully understood and are out of your control, so give careful attention to the factors that you can control.

Annual weeds compete with cereals and broadleaf crops mainly when the crops are in their earlier stages of growth. Weeds should be removed no later than 6 weeks after sowing to minimise losses. Early post-emergence control nearly always results in higher yields than treatments applied after branching in broadleaf crops.

Points to remember for the successful use of herbicides:

- Plan the operation. Check paddock sizes, tank capacities, water availability and supply.
- Do not spray outside the recommended crop growth stages, as damage may result.
- Carefully check crop and weed growth stages before deciding upon a specific post-emergent herbicide.
- Read the label. Check to make sure the chemical will do the job. Note any mixing instructions, especially when tank-mixing two chemicals.
- Follow the recommendations on the label.
- Conditions inhibiting plant cell growth, such as stress from drought, waterlogging, poor nutrition, high or low temperatures, low light intensity, disease or insect attack, or a previous herbicide application, are not conducive to maximum herbicide uptake and translocation.
- Use good quality water, preferably from a rainwater tank. Water quality is very important. Hard, dirty or muddy water can reduce the effectiveness of some herbicides.
- Use good equipment checked frequently for performance and output.
- Use sufficient water to ensure a thorough, uniform coverage regardless of the method of application.

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

- Check the boom height with spray pattern operation for full coverage of the target.
- Check the accuracy of boom width marking equipment.
- Check the wind speed. A light breeze helps herbicide penetration into crops. Do not spray when wind is strong (over 10–15 km/h).
- Do not spray if rain is imminent or when heavy dew or frost is present.
- Calculate the amount of herbicide required for each paddock and tank load. Add surfactant where recommended.
- Select the appropriate nozzle type for the application.
- Beware of compromising nozzle-types when tank-mixing herbicides with fungicides or insecticides.
- Be aware of spray conditions to avoid potential spray drift onto sensitive crops and pastures, roadways, dams, trees, watercourses or public places.
- Note that all chemicals can drift.
- 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.
- Seek advice before spraying recently released pulse varieties. They may differ in their tolerance to herbicides.
- Keep accurate spray records for each spray operation. [25]

More information

http://grdc.com.au/Media-Centre/Ground-Cover-Supplements/GCS105

6.16 Crop-topping and desiccation

With correct timing, crop-topping and desiccation can improve overall weed control and increase profitability in pulse crops. However, crop-topping is not possible in most chickpea crops.

Yield loss and grain quality impacts are severe when application timing is based on the correct ryegrass stage. However, crop-topping done when the chickpeas are ready is typically too late for preventing seed-set of the ryegrass.

Even early-maturing varieties such as Genesis™ 079 often mature too late to be safely crop-topped (see crop-topping trial data, Table 6).

The major differences between crop-topping and desiccation are:

- Herbicides used for crop-topping and desiccation are not always the same.
- Timing is not the same, as desiccation occurs after crop maturity. Crop-topping is earlier, aimed to reduce seed-set of weeds before crop maturity.
- Herbicides are registered for desiccation, as 'harvest aids', and rates used are higher than those used for crop-topping.
- Both desiccation and crop-topping will cause reduced grain quality and yield if applied at the wrong maturity stage of the crop.

See *GRDC GrowNotes (Chickpeas) Section 11: Crop desiccation.*

6.16.1 Crop-topping trials in chickpea

Key findings and comments from a South Australian Research and Development Institute (SARDI) crop-topping trial at Melton in South Australia in 2009 with chickpeas (Table 6) were as follows:

- A dry and hot November led to early senescence of pulse varieties and reduced grain yields in later maturing varieties. Many responses to the crop-topping

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

treatments may have been masked by this rapid senescence (e.g. Almaz and Genesis™114 chickpeas).

- When crop-topped 3 weeks ahead of the recommended ryegrass stage, yields of chickpeas were 17–48% of the untreated control (i.e. a yield loss of 83–52%). When crop-topped at the recommended stage, yields of chickpeas were 69–86% of the untreated control (i.e. a yield loss of 31–14%). When crop-topped 2 weeks after the optimum ryegrass stage, yields of chickpeas were 92–114% of the untreated control. Grain size was less affected by crop-topping than was yield. However, visual grain quality may have been affected to prevent delivery under national receival standards.
- Mid–late-maturing pulse varieties also showed a yield loss when crop-topped later than recommended for ryegrass control. These results indicate poor suitability of some pulse varieties and crops such as chickpea to crop-topping.
- Early-maturing chickpea lines showed yield losses from crop-topping at the recommended timing, demonstrating the difficulty in employing crop-topping to prevent ryegrass seed set in chickpeas. [26]

Table 6: Impact of timing of crop-topping on chickpea varieties of differing maturity in 2009

	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 (30 Oct.)	Plus 2 weeks (12 Nov.)		Minus 3 weeks (9 Oct.)	Recommended (30 Oct.)	Plus 2 weeks (12 Nov.)
AlmazA	1.18	19	83	92	27.4	91	92	91
PBA Slasher^A	1.96	30	70	99	15.5	87	84	100
PBA HatTrick^A	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
HowzatA	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 seeds)					18.6	16.3	15.9	18.2

Green shading denotes significant difference from the control (nil herbicide) treatment. Note: Always read the label supplied with the product before each use

Source: M. Lines and L. McMurray (SARDI), Southern Pulse Agronomy Research Trials.

6.17 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.

[26] Pulse Australia (20130 Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

- 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 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 localised 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.

6.17.1 Weed monitoring—a practical approach

Check each paddock regularly and often enough to identify critical stages of crop and weed development for timely intervention, and to evaluate efficacy of weed-management practices. This weed scouting can be done whenever you search for insect pests and beneficials, or enter the paddock to plant, tend, irrigate or harvest crops. Inspect for weeds every few days during crop germination, emergence and early establishment. Later, checking once a week is usually sufficient.

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 or pH 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
- 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?

6.18 Mode of action

6.18.1 Mode of action matters

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.

6.18.2 Mode-of-action labelling in Australia

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. However, six new herbicide MOA groups were created to group herbicides more accurately.

More information

Refer to the APVMA website to obtain a complete list of registered products from the PUBCRIS database: www.apvma.gov.au

Refer to the APVMA website to obtain a complete list of registered products from the PUBCRIS database: www.apvma.gov.au.

6.19 Further reading

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SECTION 7

Insect control

7.1 Insect pest management

Insect pest management in pulses is more than just chemical control. Correct identification of the pest or beneficial insects is critical.

Farming practices have a major impact on insect pest incidence and control needs. For example, aphids are worse in bare-ground situations or in early sowing when summer weeds are present. [1]

7.2 Insect control thresholds

Insect control thresholds provide guidelines to allow timely decisions for crop spraying. This can reduce unnecessary spraying and keep populations from reaching a level where damage is high.

The most common threshold used is an economic threshold, which involves control at a density that will prevent pest numbers from reaching an economically damaging population.

The aim of pest management is to keep pest populations below this economic threshold.

Guideline thresholds based on research exist for some pests but most thresholds fluctuate depending on a number of factors. Monitoring and sampling of crops are essential to determine these factors and their influence on where the threshold lies. Farmers who maintain a close watch on pest activity through regular crop inspections and thorough sampling are in a better position to decide if, and when, treatment is needed.

The following factors should be monitored and considered when using thresholds and making spray decisions:

- environmental conditions and the health of the crop
- extent and severity of the infestation and how quickly the population increases
- prevalence of natural control agents such as parasitic wasps, predatory shield bugs, ladybirds and diseases
- type and location of pest damage and whether it affects yield indirectly or directly
- stage in the life cycle of the pest and the potential for damage
- crop stage and ability of the crop to compensate for damage
- amount of damage that has already occurred and the additional damage if the crop is not sprayed
- value of the crop (high value crops cannot sustain too much damage as a small loss in yield or quality could mean a large financial loss), the cost of the spray and its application, and the likely yield or quality benefit gained from control [2]

More information

http://thebeatsheet.com.au/wp-content/uploads/2012/04/GoodBadBug-FINALscreen22Feb3.pdf

http://thebeatsheet.com.au/about/

http://thebeatsheet.com.au/helicoverpa/an-economic-threshold-calculator-for-helicoverpa-chickpeas/

http://ipmworkshops.com.au/wp-content/uploads/Chickpea_IPM-Workshops_north-March2013.pdf

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

[2] Pulse Australia (20130 Northern chickpea best management practices training course manual—2013. Pulse Australia Limited.

7.2.1 Beneficial insects

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.

If chickpeas are poorly managed, they can contribute large numbers of *Helicoverpa* to the local populations, posing a threat to susceptible summer crops (sorghum, pulses, cotton) grown in the district. To manage *Helicoverpa* well, it is important to sample and identify the different larval instars (very small, small, medium–large, large). Familiarity with these different life stages is critical to determining the likelihood of damage and optimising the timing of control.

Two species of *Helicoverpa*, *H. armigera* (the corn earworm or cotton bollworm) and *H. punctigera* (the native budworm), may occur in chickpea in the northern region. *Helicoverpa armigera* is resistant to some insecticide groups (particularly the synthetic pyrethroids), whereas *H. punctigera* is susceptible to all products. Although it is not always possible to do so, identifying which species is present, or knowing which predominates in the local area, may help growers avoid products that give insufficient control. Some tools can help growers to make this determination. [3]

More information

http://thebeatsheet.com.au/helicoverpa/an-economic-threshold-calculator-for-helicoverpa-chickpeas/

http://ipmworkshops.com.au/wp-content/uploads/Chickpea_IPM-Workshops_north-March2013.pdf

7.3 Pest-management process

Figure 1 outlines the steps in the pest-management process.

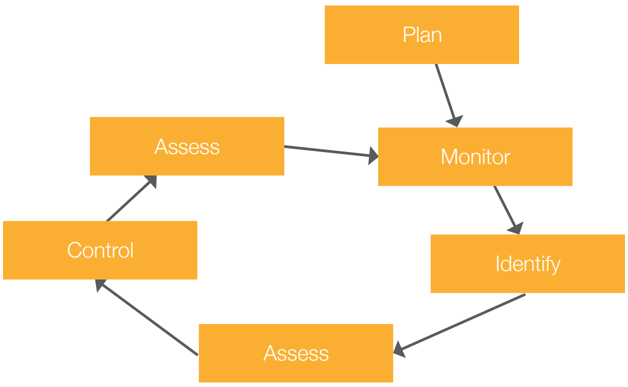

Figure 1: The pest management process.

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.

[3] M Miles (2013) Chickpea insect pest management. Northern grains region. Department of Agriculture, Fisheries and Forestry Queensland, http://ipmworkshops.com.au/wp-content/uploads/Chickpea_IPM-Workshops_north-March2013.pdf

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). [4]

7.4 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.

7.4.1 Registration

All pesticides go through a process called registration, where they are formally authorised (registered) by APVMA for use:

- against specific pests

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

- 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

7.4.2 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 are also essential reading. These document the hazards posed by the product, and the necessary and legally enforceable handling and storage safety protocols.

More information

Details of product registrations and permits are available via the APVMA's website: www.apvma.gov.au.

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.

APVMA

The national body in charge of administering these processes is APVMA and is based in Canberra.

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. [5]

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

7.5 Native budworm (*Heliothis*) (*Helicoverpa* spp.)

Helicoverpa spp. are the major insect pests of chickpeas. Other less frequent pests include locusts, aphids, cutworms, false wireworms and blue oat mites.

The larvae of *Helicoverpa* are the main insect pest of chickpeas. It is important to be able to identify the different larval instars (very small, small, medium, large) of *Helicoverpa*. If possible, identifying which of the two species (*H. armigera* or *H. punctigera*) is present, or knowing which is predominate in your area, may help to avoid products that may not provide adequate control.

Description

Figure 2: Native budworm (Helicoverpa, previously Heliothis) moths, showing male (right) and female (left). Note the buff colouring. (Photo: SARDI)

Adult moths are nocturnal, so are rarely seen during the day. They vary in colour from grey-green to pale cream and have a wingspan of 3–4.5 cm. The hind wings have a dark, broad band on the outer margin (Figure 2).

Adult moths lay round eggs (0.5 mm in diameter) singly on the host plant. The eggs are white but turn brown just before hatching. The larvae grow to 5 cm long and vary in colour from green, yellow pink and reddish brown to almost black (Figures 3 and 4).

Figure 3: Left to right: fresh white, brown ring and black larval head in nearly hatching eggs.

Figure 4: Helicoverpa larvae occur in a range of colours.

Figure 5: In spring, eggs hatch in 1–2 weeks and larvae feed for 4–6 weeks. This shows all stages from egg to fully grown larvae. Insecticides are more effective on smaller larvae. (Photo: SARDI)

Larvae can be easily identified, despite the colour variation, by a broad yellow stripe along the body (Figure 5). The young larvae (<10 mm) prefer to feed on foliage. Older larvae prefer to feed on pods.

Other larvae, which look like native budworm (*H. punctigera*), may be found in a pulse crop (e.g. southern armyworm and pink cutworm). These are primarily grass feeders and rarely do any damage to pulses.

Two species of Helicoverpa

Both *H. armigera* and *H. punctigera* are found in chickpea in Australia and attack a wide variety of crops, including chickpea.

Visual identification of the different species is sometimes possible from examination of larvae. Small *H. armigera* larvae (3rd instar) have a saddle on the fourth segment and *H. punctigera* do not. This is often difficult to see in the field and the method is not 100% reliable, but may be used as a guide.

In larger (5th and 6th instar) larvae, hair colour on the segment immediately behind the head is a good indicator of species. These hairs are white in *H. armigera* and black in *H. punctigera* (Figure 6). Moths can be differentiated by hindwing colour pattern (Figure 7).

Figure 6: Head of 5th instar *H. armigera* larva showing the white hairs on the segment immediately behind the head. Larvae of *H. punctigera* have black hairs. (Photo: R. Lloyd, DPI&F)

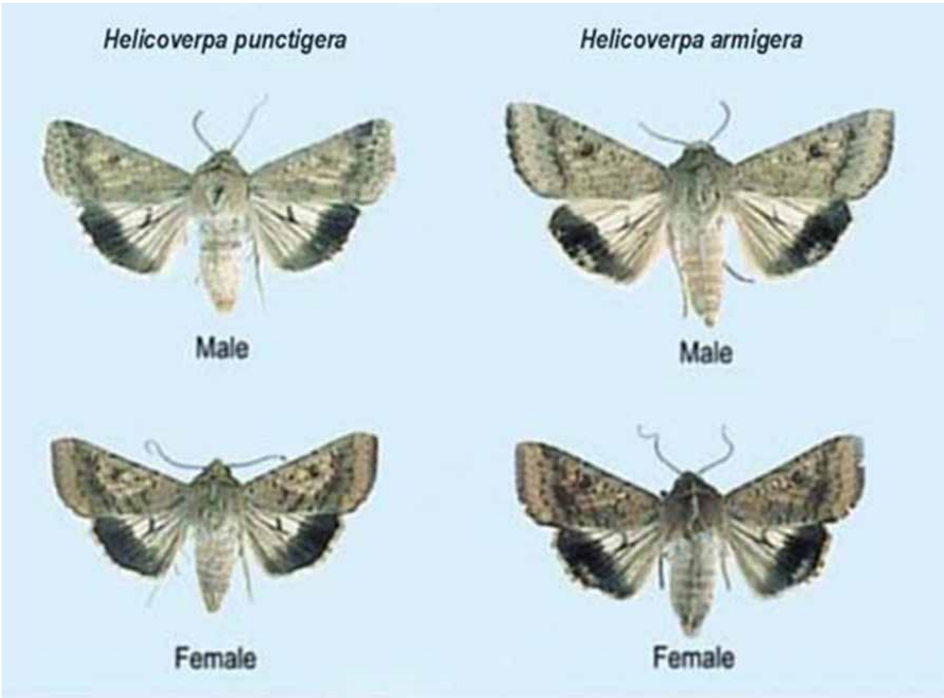

Figure 7: *Helicoverpa punctigera* and *H. armigera* moths are distinguished by the presence of a pale patch in the hindwing of *H. armigera*.

Species composition can vary and will affect control decisions

Species composition in the crop will be influenced by the time of year. In temperate regions (southern Queensland and further south), the majority of *H. armigera* individuals over-winter from mid-March onwards and emerge during September–October. *Helicoverpa punctigera* is usually the dominant species through September, but seasonal variation can lead to *H. armigera*-dominant early infestations in some years, particularly in more northern districts. Pheromone trap catches can be used as an indication of the species present in a region, although they are not a reliable predictor of egg lay within a crop.

If the level of *H. punctigera* infestation is high, any registered product will control the larvae. If *H. armigera* is the dominant species, spray failures with carbamates or pyrethroids may occur because of resistance. The biopesticides *Helicoverpa* nucleopolyhedrovirus (NPV) and *Bacillus thuringiensis* (Bt) currently have no known resistance problems. [6]

7.6 Why distinguish the two species of *Helicoverpa*?

Determining which *Helicoverpa* species is present in the crop is essential, principally because of the differing susceptibility of the two species to synthetic pyrethroids and carbamates.

Visual identification of the different species is sometimes possible from examination of larvae; however, this can be difficult and unreliable with small larvae about the size when control decisions have to be made. A hand lens, microscope or USB microscope is essential for examining small larvae.

Small *H. armigera* larvae (3rd instar) have a saddle on the fourth segment, whereas *H. punctigera* do not (Figure 8). In larger (5th and 6th instar) larvae, look at the hair colour on the segment immediately behind the head—white for *H. armigera* and black for *H. punctigera* (Figure 9).

Figure 8: Medium Helicoverpa armigera (12 mm) showing the distinctive 'saddle' on 4th and 5th body segments (top), and H. punctigera without saddle (bottom).

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

Figure 9: Large *Helicoverpa punctigera* (left) and *H. armigera* (right) larvae showing the distinguishing dark and pale hairs, respectively, behind their heads.

7.6.1 Species composition can vary between seasons and regions

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* from inland Australia to eastern cropping regions.
- Winter rainfall in eastern cropping regions, which drives the abundance of local populations of *H. armigera* through the generation of spring hosts. In regions where chickpeas are grown, they may serve as a significant spring host for *H. armigera* emerging from diapause if these populations are not controlled (e.g. sub-threshold populations across large areas of chickpea, or poorly managed crops).
- Relative timing of flowering–podding (attractive and susceptible) stages and the immigration of *H. punctigera* and emergence of *H. armigera* from overwintering diapause. Note that in Central Queensland, *H. armigera* does not enter winter diapause and will be the predominant species in chickpeas.
- Geographic location. In temperate regions (southern Queensland and further south), 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, particularly in more northern districts. Pheromone trap catches can be used as an indication of the species present in a region. Note that pheromone traps cannot be used to predict the size of an egg-lay within a crop. [7]

7.7 Monitoring chickpeas for insect pests

Chickpeas are susceptible to significant yield loss caused by *Helicoverpa* from podset through to harvest. Although *Helicoverpa* can cause reductions in both yield and quality,

[7] 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

the economic threshold for minimising yield loss is much lower than that which would result in a reduction in grain quality.

Seedling insect pests such as cutworm can attack chickpeas, but are rarely an economic problem. Other infrequent pests include blue oat mites, false wireworms, and aphids.

Sampling with a beat sheet is best practice for monitoring *Helicoverpa* in chickpeas. Establishment pests will be detected by visual inspection of seedlings.

Regular monitoring during the susceptible crop stages is critical, particularly for *Helicoverpa*, where you may be dealing with insecticide-resistant larvae, and good control depends on targeting of small larvae.

7.7.1 Sampling strategy and technique

The beat sheet and sweep net are accepted techniques for monitoring *Helicoverpa* larvae in chickpeas. The beat sheet is the recommended technique for crops grown on wider row spacings (>50 cm rows) (Figure 10).

Economic thresholds are developed using a specific sampling technique, an important consideration when making management decisions.

Figure 10: The beat sheet is the recommended technique for sampling crops grown on wider spacings.

Beat sheet v. sweep net

The sweep net and beat sheet have not been calibrated in chickpea, so it is not yet possible to use the one threshold with an adjustment for relative sampling efficiency of the sweep net.

Using a beat sheet

Check crops regularly (at least once a week) from flowering through to harvest, using a beat sheet. In addition to larval counts, visual observation of the crop growth stage, progress of flowering and podding, and the presence of eggs, diseased larvae (NPV) and moths all provide useful information for decision-making.

Each time you inspect, check at least five 1-metre sections of row at a number of sites in the field. Start sampling at least 50 m into the field, and include samples from well into the field to enable a representative average field population to be calculated.

Video

Watch a video of sampling chickpeas with a beat sheet at http://www.youtube.com/user/TheBeatsheet

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.

Video

Watch a video of a sweep net being used to sample barley for armyworm at http://www.youtube.com/user/TheBeatsheet

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 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.

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
- management recommendation (economic threshold calculation)
- post spray counts

The *Helicoverpa* size chart (Figure 11) 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.

Helicoverpa larval size categories and actual sizes

Actual larval size	Larval length (mm)	Size category
	1-2mm	very small
	4-7	small
	8-23	medium
	24-30+	large

Figure 11: Helicoverpa larval size categories and actual sizes.

Eggs

There are no egg thresholds in chickpeas. Relying solely on egg counts for control decisions in chickpea is unreliable. This is largely due to the difficultly in accurately detecting eggs on the chickpea plant. Egg survival through to larvae can also be highly variable; therefore, decisions based on egg numbers are less accurate than those based on larval density.

Instead, use egg counts as an indication of an egg-lay event and determine the potential development rate of the *Helicoverpa*. Continue sampling to target small larvae for control.

Eggs take 2–5 days to hatch (generally slower in winter than summer due to temperature differences). Newly laid eggs are white. They turn a brown or off-white colour after a day or two. Eggs very close to hatching are in the 'black head' stage, where the egg is darker in colour and it is possible to see the black head capsule of the developing larva inside.

Very small larvae

The numbers of very small larvae, those that have hatched from the egg in the previous 24 hours, are difficult to estimate in the field. Often they are low in the canopy and remain on leaflets when they fall onto the beat sheet, making them very difficult to see and count. Very small larvae do no economic damage to the crop, their feeding confined to leaves. Early research on *Helicoverpa* showed that the mortality of very small larvae is very high and their value in chickpea monitoring is most likely as indicators of an egg-lay and of potential activity of larger larvae in a week or two. A more detailed discussion follows of why counting and recording very small larvae does not contribute greatly to decision making. [8]

More information

See picture on page 14 of 'Insect Ute Guide (Northern Grain Belt)': http://www.grdc.com.au/Resources/Ute-Guides

https://itunes.apple.com/au/app/insect-id-ute-guide-tablet/id667854493?mt=8

7.8 Economic thresholds for *Helicoverpa*—the cornerstone of decision-making

The economic threshold is classically defined as the pest population likely to cause a loss of yield and/or quality equal in value to the cost of control (chemical plus application). At threshold, the impact of the pest is equivalent to the cost of control, so there will be an economic benefit from controlling the population only if it exceeds the economic threshold.

Calculation of an economic threshold is based on four factors: (*i*) the cost of control, (*ii*) crop value, (*iii*) the average number of insect pests per sampling unit, and (*iv*) the potential loss per pest insect.

7.8.1 Economic thresholds for *Helicoverpa*

This economic threshold is based on beat-sheet sampling (DAFF Queensland 2007).

Vegetative to early flowering: High populations have no impact on yield or quality. In rare situations, control may be warranted during the vegetative and flowering stages, when pest pressure is extreme and plants are defoliated.

Mid-flowering to early podding: Value of crop loss is calculated by the following equation (or refer to the ready-reckoner, Table 1):

Yield loss ($/ha) = {no. of *Helicoverpa* larvae per m^2 x 2.0 x chickpea price ($/t)}/100

where 2.0 represents 2 g grain consumed per larva.

This equation has been used to produce the ready-reckoner table (Table 1) 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.

8 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

Table 1: 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

Control is warranted if the cost of control is less than the value of the yield loss predicted [9]

Chickpea price ($/t)	1 larva/m²	2 larva/m²	3 larva/m²	4 larva/m²	5 larva/m²
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

To calculate a ready-reckoner for economic thresholds (larval density) (Table 2), rather than the value of yield loss, use the following formula:

Economic threshold (no. of larvae/sampling unit) = C ÷ (D x V)

where C is cost of control ($/ha), D is yield loss per larva per sampling unit (kg/ha), and V is chickpea price ($/t).

Table 2: Economic threshold (no. of larvae per sampling unit) ready-reckoner

Assume D = 20 kg/ha (0.02 t/ha)

Cost of control ($/ha)	Crop value ($/t)							
	200	250	300	350	400	450	500	550
10	2.5	2.0	1.7	1.4	1.3	1.1	1.0	0.9
15	3.8	3.0	2.5	2.1	1.9	1.7	1.5	1.4
20	5.0	4.0	3.3	2.9	2.5	2.2	2.0	1.8
25	6.3	5.0	4.2	3.6	3.1	2.8	2.5	2.3
30	7.5	6.0	5.0	4.3	3.8	3.3	3.0	2.7
35	8.8	7.0	5.8	5.0	4.4	3.9	3.5	3.2
40	10.0	8.0	6.7	5.7	5.0	4.4	4.0	3.6

7.8.2 Dynamic economic thresholds for the northern region

Recently, there have been changes to the way we discuss economic thresholds for *Helicoverpa* (beat sheet based threshold). The most obvious change is that it is no longer a set number of larvae per m². Rather, it is dynamic, and is responsive to the value of the crop ($/t) and the cost of control ($/ha).

Research by Department of Agriculture, Fisheries, and Forestry (DAFF), Queensland, entomologists has examined the impact of *Helicoverpa* on chickpea yield and quality. As a result of this work, the potential yield and quality loss that larvae will cause under average field conditions has been quantified.

The research in Queensland determined the consumption rate of the larvae from hatchling to pupation to be 20 kg of grain per ha per larva per m² (the outcome of the relationship between larval feeding and the compensation by the chickpea plant). Loss attributed to a particular larval density is calculated on the basis that they are large larvae doing maximum damage. The larval density in the relationship was determined using a beat sheet, and the thresholds are accurate only for larval density estimates made using a beat sheet.

Making a decision about whether it is economic to spray, based on specific parameters for each management unit (field), is now possible. Following on from this, a benefit/cost ratio can be applied to the crop. This allows the farmer to determine the risk level. For example, if potential damage to crop is $30/ha, and control cost is $20/ha, there is a

9 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

ratio of 1.5. Depending upon the individual farmer, they may believe this ratio warrants action, or they may prefer to wait until a ratio of 2 occurs to justify action.

7.8.3 Additional information on using the threshold to make a decision

Helicoverpa activity during flowering does not result in yield loss

Data from six trials in 2006 (DAFF, Queensland) clearly show that controlling *Helicoverpa* at flowering does not result in a significant increase in yield or quality over delaying control until podset (expanded pods) (Figure 12).

In rare situations, control may be warranted during the vegetative and flowering stages, when pest pressure is extreme. If using products that are effective only against small larvae, it may be necessary to apply a spray during flowering to control a population of small larvae to prevent them from causing damage as large larvae during podset and pod-fill.

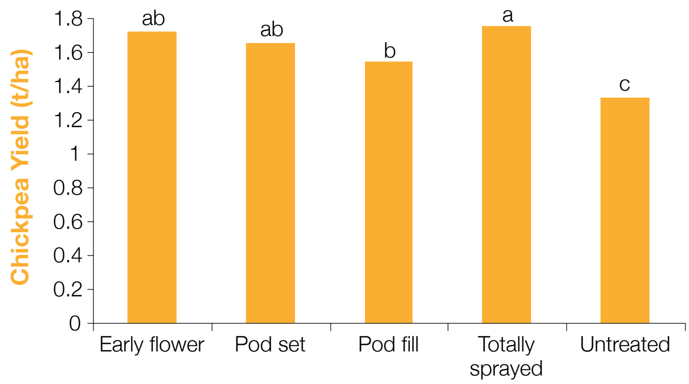

Figure 12: The impact of Helicoverpa on chickpea yield when controlled at different stages of crop maturity. Bars with the same letter are not significantly different from each other.

Larval feeding behaviour—how this influences crop damage

Extensive observational studies of small and large larvae on chickpeas demonstrate that small larvae are primarily foliage feeders, whereas large larvae feed on pods and foliage (see Figure 13). Neither small nor large larvae were observed to have any preference for flowers, which supports the data showing no yield loss when *Helicoverpa* larvae are tolerated during flowering.

If using a product that is effective only against early-instar larvae (e.g. NPV), then the application of control may be necessary during flowering to prevent damage by large larvae at podset.

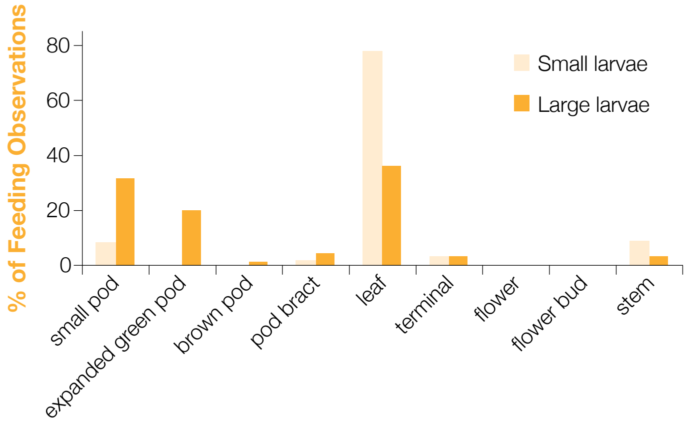

Figure 13: Feeding preferences of small and large Helicoverpa larvae on chickpea when allowed to select feeding sites over a 4–6-h period. Small larvae were 3rd instar, large larvae were 4th–5th instar.

Grain quality

Grain quality is not affected in the range of larval densities for which it is currently economic to spray to prevent yield loss. Recent trials have shown that in the range of 1–4 larvae/m^2, defective grain is well below the level at which discounts or penalties apply (6% by weight; National Agricultural Commodities Marketing Association (NACMA) 2006). Similarly, in the time-of-spraying trials, there was no significant decline in grain quality other than in the untreated plots (Figure 14). Given this result, we can be confident that within the range of larval densities for which it is economic to control, and with the recommendation to withhold treatment until late flowering or podset, quality loss does not need to be factored into the economic threshold.

There is anecdotal information suggesting that larvae will cause significantly more pod damage in exceptional circumstances (e.g. extremely high temperatures, extreme crop moisture stress); however, no trial work had been done to determine to verify or quantify this.

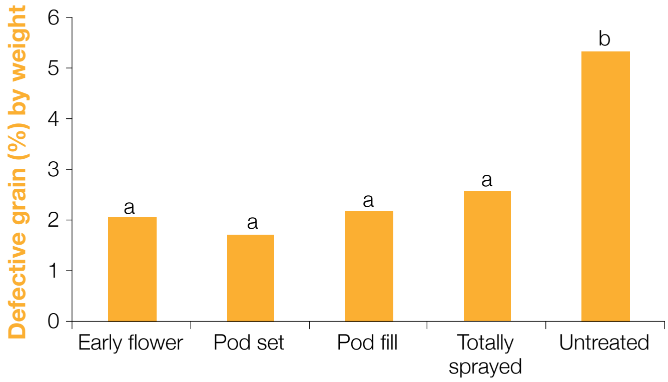

Figure 14: Proportion of defective grain (% by weight) did not increase when Helicoverpa larvae were tolerated during flowering. Bars followed by the same letter are not significantly different from each other.

Natural mortality of larvae is high

The number of large larvae recorded in unsprayed situations is always considerably lower than the number of small larvae recorded in earlier checks. This indicates significant mortality of small larvae. Recent trial work at 19 sites from central Queensland to the Darling Downs has shown an average loss of 70% of larvae from small to large in untreated plots (Figure 15). That is, only one-third of the small larvae recorded survived to become large. In previous studies of *Helicoverpa* larval feeding behaviour, large larvae were found to cause most of the damage, about 80%, with medium larvae contributing about 15% and other instars the remainder (Figure 16). A similar result is expected for chickpea.

This natural mortality (most likely a result of dislodgement from the plant, disease, cannibalism) is important because it means that a proportion of the population will not survive to cause damage to the crop, even if there is no control. Conservatively, it is estimated that the number of small larvae (<7 mm) found in a field sample can be adjusted for natural mortality by assuming that 30% will not survive to cause significant damage as medium and large larvae. Estimating the number of very small larvae is difficult and unreliable in the field, and the inclusion of these larvae in estimates of the population is potentially misleading. Although the data show overall an average 70% mortality across a large number of fields, the average is calculated from a range of mortality from 1% through to 99%; therefore, mortality will vary considerably from field to field. Hence, the conservative suggestion of 30% mortality adjustment rather than 70% adjustment.

In practice, this means that when calculating the number of larvae per m^2 in a field, the following equation is used:

Number per m^2 = {(S x 0.7) + M + L}/row spacing (m)

where S is small, M is medium and L is large larvae; number of larvae is based on the number per metre of row, assessed with a beat sheet; and the multiplier 0.7 is 70% surviving population. Dividing by the row spacing (in metres) adjusts the density for different row spacing.

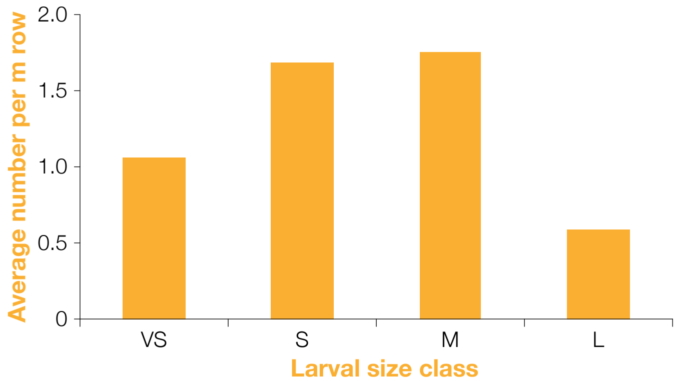

Figure 15: Average numbers of Helicoverpa larvae in the different age classes in untreated chickpea. Data are averaged across 19 sites across central Queensland and the Darling Downs.

Figure 16: Large Helicoverpa larva feeding on a chickpea pod.

Knowledge gaps

There are no data on the behaviour of larvae in extremely moisture-stressed crops *v.* crops with adequate soil moisture. This means that we cannot say whether there is more, or earlier, flower and pod feeding when foliage appears to be less attractive.

Likewise, there are no data comparing the feeding behaviour of *H. armigera* and *H. punctigera*. The question remains as to whether the different species have differing preferences for leaves, flowers and pods. [10]

7.9 Use of *Helicoverpa* economic thresholds—an example

Site: Camerons
Date: 15/9/06
Row spacing: 75cm

Sample (1 m row beat)	VS	S	M	L
1	8	5	1	0
2	1	1	1	0
3	3	3	0	1
4	3	2	1	0
5	2	6	0	0
Average		3.4	0.6	0.2
Adjust for 30% mortality (S*0.7)	(3.4*0.7)=2.4			
Mean estimate of larval number (Adjusted S)+M+L	2.4 0.6 0.2 = 3.2			

Adjust for row spacing
divide by row spacing (m) $\frac{3.2}{0.75}$

4.2 Density Estimate per square metre

Figure 17: Example of a field check sheet with sampling data recorded for Helicoverpa larvae in chickpea.

In Figure 17, 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 1) 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. [11]

7.10 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. armigera* and *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.

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10 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

11 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

7.10.1 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 (Figure 18). This is particularly so for *H. armigera*.

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.

Figure 18: Helicoverpa larva inside a chickpea pod. Larvae that are not controlled when they are small can cause major crop damage.

7.10.2 Key considerations

Chickpeas provide the first host for H. armigera each season

Traditionally, control options have been carbamates and pyrethroids. Resistance to pyrethroids is generally high and spray failures may occur if the population is predominantly *H. armigera*. Recent seasons have seen carbamates perform more reliably as their use in other crops (e.g. cotton) has declined. *Helicoverpa punctigera* is currently susceptible to all chemical groups. Both species are currently susceptible to indoxacarb and the biopesticides, NPV and Bt.

Beneficial insects make little contribution to Helicoverpa control in chickpeas

Malic acid on the chickpea leaves has a repellent effect on many species, especially wasps such as *Trichogramma* and *Microplitis*. The group of parasitic flies called tachinids has been recorded parasitising *Helicoverpa* in chickpeas. The tachinid fly lays its eggs on the larva, usually near the head capsule (see pages 124–125 in the Northern region Ute Guide, and DPI&F brochure 2005: Parasitoids: Natural enemies of helicoverpa). The tachinid larva then feeds and develops inside the *Helicoverpa* larva, and the adult fly emerges from a cocoon in the mature larva or pupae. Tachinid flies usually kill late-instar larvae; therefore, larvae will cause crop damage before they die. For this reason, tachinids have no effect on the damage that larvae can do to a crop, and their presence does not influence the estimate of larval numbers in the crop.

More information

http://www.croplifeaustralia.org.au/default.asp?V_DOC_ID=1952

Resistance management underpins product efficacy

The threat of resistance development to new products will influence their future use patterns. It is likely that chemical companies will avoid season-long use of a single product in a sequence of crops to minimise the likelihood of resistance developing.

The Australian cotton industry has a voluntary Insecticide Resistance Management Strategy (IRMS) that incorporates all insecticides registered for use in cotton. The grains industry does not have an IRMS, although CropLife has strategies for insecticide resistance management for a number of crops

7.10.3 Steward™ (indoxacarb) and the IRMS

Until 2012, indoxacarb use in chickpeas was restricted to avoid lengthy exposure of *Helicoverpa* in the farming system—cutoff 15 October for warm areas, September 15 for hot areas, and October 30 for cool areas.

This restriction has now been lifted, as use patterns in chickpea are, in most seasons, consistent with the recommended window for each region. [12]

7.11 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 podset begins. This situation potentially leads to high numbers of advanced stage larvae, resulting in more costly and less reliable control.
- 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.

The presence of *H. armigera* will influence management decisions. The *Helicoverpa* emergence model is available through the COTTASSIST website: https://www.cottassist.com.au/DIET/about.aspx.

The use of pheromone traps in spring and close visual inspection of larvae provide information on the likely presence of *H. armigera* in chickpeas as the season progresses.

Biopesticides (NPV and Bt) must target smaller larvae (preferably <7 mm in length). Therefore, in situations with high larval densities or across a range of size classes, biopesticides are not the preferred choice. For more information, see the DAFF Queensland brochure 'Using NPV in field crops': http://thebeatsheet.com.au/resources/.

- Keep pod damage in perspective. While larvae and their damage may be evident in a crop, counts of damaged and undamaged pods will give an estimate of actual yield loss accumulating (1 damaged pod per m^2 = 1.7 kg/ha of yield loss). In most

12 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

cases relative pod damage is far less than initial visual inspections suggest, so careful monitoring of damage in relation to total pod load is recommended. Once pods are damaged, the yield is already lost; pod damage is not useful in making control decisions to prevent yield loss.

- Be aware of withholding periods. Both Steward™ and Larvin® have long withholding periods (21 days), as do some other products. Be aware that late sprays of these products could delay the harvest date.
- Resistance management is vital. The grains industry relies heavily on a limited number of effective insecticides for *Helicoverpa*, particularly for *H. armigera*. Abiding by key insecticide resistance management strategies is good practice:
- Target small larvae to maximise efficacy (Figure 19).
- Be aware of the probability that the population will contain *H. armigera* and select insecticides accordingly.
- If more than one application is made in a crop, rotate insecticide groups.
- Where spray failures are suspected, do not re-treat with the same product.
- Check compatibility of insecticides with fungicides if planning to use together. Mixing fungicides with insecticides is becoming more common because of the fungicide spray programs recommended for control of Ascochyta blight. Some product formulations are not compatible with available fungicides. [13]

Figure 19: Small Helicoverpa larva feeding on exposed sites and vegetative growth as pictured are easily targeted with sprays. (Photo: R. Lloyd, DPI&F)

7.12 Area-wide management strategies for *Helicoverpa*—the role of chickpeas

When assessing whether to control *Helicoverpa* in a chickpea crop, the decision is usually based only on potential yield loss in that crop.

Reduction of the overall size of the *Helicoverpa* population on a regional basis is the aim of an area-wide management (AWM) strategy. It is a move from the paddock-by-paddock control of *Helicoverpa* to an approach where neighbours and their agronomists communicate and cooperate to reduce numbers wherever they can.

Tactics that may be considered in the context of an AWM approach and relate to *Helicoverpa* management in chickpea include:

- reducing the spring *H. armigera* generation in commercial chickpea crops, and minimising the carryover of moths from chickpeas to susceptible summer crops

13 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

- insecticide resistance management

Chickpeas can host the first generation of Helicoverpa armigera

It is well recognised that chickpeas provide a host for the first generation of *Helicoverpa* in spring. *Helicoverpa armigera* emerge from diapause from late September to October. Temperature variations in temperate regions will mean a variation in emergence from year to year, and region to region; the warmer the temperatures, the earlier *Helicoverpa* moths will emerge. Commercial chickpea crops that are still at flowering and pod-fill stages will be attractive to *Helicoverpa*.

Most larvae developing during normal pod-fill will emerge as moths before harvest, so pupae-busting of these crops is of little benefit. Where there are late larval infestations, cultivation soon after harvest will kill many pupae. Using larvae-checking records and the estimates of *Helicoverpa* development (see Appendix 1), it is possible to estimate the time-frame for effective pupae-busting. Left too long, the pupae will emerge as moths and move into other crops.

These strategies will help to reduce the buildup of the first generation of *Helicoverpa* within a farming region. [14]

7.13 Control options

Choosing the most appropriate chemical for pest control will depend on which pests are present (Table 3), in what numbers, their stage of development (whether eggs, larvae/nymphs or both), resistance levels, and location in the crop (i.e. in flowers or leaf feeding).

Effective insecticides need to be used in a way that will not increase the risk of developing resistance or increasing resistance to these products in the target species or other pests.

Key points:

- Use economic thresholds to make spray decisions.
- Be aware of, and use, the voluntary farming systems IRMS.
- Avoid prolonged use and over-reliance on any one chemical group for *Helicoverpa* control.
- Rotate the main chemical groups where possible.
- Avoid use of pyrethroids on *H. armigera* populations where possible.
- Check compatibility of potential mixing partners before recommending and applying.
- Always read the label supplied with each product before use.
- Abide by withholding periods and factor this into your decision-making about harvest date and/or insecticide use.
- Target small insects.

7.13.1 Resistance management and product selection

It is unlikely that season-long use of a single product will be allowed. For some products, registration on grain crops may not be considered, because of selection pressure, resistance threat, residues and cost.

An IRMS that incorporates all insecticides and all crops in the farming system is required. It allows the best placement for these new products that satisfies sustainable management approaches, and viable commercial returns to agrochemical companies.

14 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

7.13.2 Other management considerations

Some products (including Steward™ and Larvin®) have long withholding periods. Late sprays of these products could delay the harvest date.

Determine the *Helicoverpa* species present. Consider consulting with entomologists and other agronomists on which products are working in each region. Where spray failures are reported, consider the range of possible causes, including resistance. For example, poor application (coverage, timing) can be mistaken for resistance. [15]

7.14 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.

7.14.1 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 3 lists several commonly used insecticides and highlights some known incompatibilities with mancozeb. Table 4 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. [16]

Always ensure that a product (or mixture) is safe for the crop before recommending and applying.

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

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

Table 3: Compatibilities of various insecticides with mancozeb

Product	Mancozeb compatibility	Considerations
Larvin 80 WG	Yes	
Larvin 375	Dithane DF, no	
	Dithane M45, yes	Use within 6 h of mixing
Gemstar	Dithane DF, yes	Check pH is within the range 7–8.5
	Dithane M45, yes	Check pH is within range 7–8.5
Lannate EC	Yes	Mix mancozeb first, Lannate second. Use as soon as possible after mixing
DiPel SC	Yes	Mix fungicide first and check pH. A pH >8.5 will require the addition of a buffer to reduce the pH
Karate EC	Yes	Mix mancozeb first, Karate second. Use as soon as possible after mixing
Karate ULV	No	

Table 4: Compatibilities of various insecticides with chlorothalonil

Product	Chlorothalonil compatibility	Considerations
Steward™ (indoxacarb)	Yes. Widely used with chlorothalonil and no known 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 powder formulations of the most commonly used fungicides, insecticides 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-injurious to your crop. This product should not be mixed with spraying oils or sprayed onto 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 plant injury

Source: above tables compiled with the assistance of Bayer CropScience, Sumitomo Chemical Australia, DuPont, Crop Care Australasia and Infopest.

7.15 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

More information

https://grdc.com.au/Resources/Bookshop/2015/02/Crop-Aphids-Back-Pocket-Guide

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. [17]

7.16 Blue-green aphid *(Acyrthosiphon kondoi)*

The malic acid in chickpeas means that there is little colonisation of chickpea plants by blue-green aphids (Figures 20 and 21). However, blue-green aphids transmit Cucumber mosaic virus (CMV), a non-persistent virus, when visiting chickpea crops. Therefore, management of chickpea crops for protection against blue-green aphid invasion from surrounding areas is important.

Figure 20: Close up of blue-green aphid. (Photo: Grain Legume Handbook)

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

Figure 21: Blue-green aphids. Note the young and older aphids. The brown aphids are dead blue-green aphids that have been parasitised by wasps. (Photo: Grain Legume Handbook)

Lifecycle

Blue-green aphids prefer cooler weather (10–18°C) for breeding. Females produce up to 100 young at a rate of about 7 per day. Winged aphids develop when infestations become crowded. Winged aphids fly or are blown by the wind to start new infestations elsewhere.

Monitoring

Monitor in chickpea and surrounding crops at all crop stages. Colonisation of chickpeas does not occur as it does in other pulses (e.g. lentil or lupin), so invasion comes from adjacent crops and pastures.

Frequent monitoring of seedlings and establishing plants is necessary to detect rapid increases of aphid populations. Stem samples give useful estimates of aphid density. When damage is apparent, intervention may be necessary. Moderating factors include the degree of aphid tolerance of the crop (chickpea suffers little mechanical damage), availability of moisture and the incidence of predators and parasitoids.

Control

In other pulses, a neonicotinoid (Gaucho® 350SD) insecticide seed dressing could be used on susceptible crops to prevent aphids from attacking emerging plants and spreading the persistently transmitted viruses *Bean leaf roll virus* (BLRV) and *Beet western yellows virus* (BWYV) early in the season. Note, though, that Gaucho® (imidacloprid) is not registered in chickpea.

The best protection against aphid infestation and virus spread in chickpea is to control the aphid population and its hosts beforehand. A prophylactic insecticide spray in chickpea to prevent aphid incursion is not desirable.

Spray with insecticide only where damage to growing points is obvious. Broad-spectrum insecticides should be avoided, for conservation of natural enemies.

More information

For additional aphid and virus control details see Pulse Australia Bulletin 'Virus management': www.pulseaus.com.au

If blue-green aphid 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.

Natural enemies are: hoverfly larvae, aphid parasites, green lacewing larvae, brown lacewing and ladybirds. [18]

7.17 Cowpea aphid *(Aphis cracciuora)*

The malic acid means that there is little colonisation of chickpea plants by cowpea aphids (Figure 22). However, cowpea aphids transmit the non-persistent virus CMV and the persistent virus BWYV when visiting chickpea crops. Management of chickpea crops for protection against cowpea aphid invasion from surrounding areas is therefore important.

Figure 22: Cowpea aphids. Note the different aphid ages, young to old. The older aphids are shiny black. The white cast is a skin, shed as the aphid grows. (Photo: Grain Legume Handbook)

Lifecycle

An infestation of cowpea aphids is generally patchy at first but they will spread through the crop if the weather is fine and warm. Infestations start when winged females colonise a few plants in a crop and give birth to wingless nymphs that live in colonies. This may occur from early winter onwards. As the plant deteriorates the aphids move to neighbouring plants, and the area of infested patches within the crop increases.

Monitoring

Monitor for cowpea aphids in chickpea and surrounding crops and pastures at all crop stages. Colonisation of chickpeas does not occur as it does in other pulses, so invasion comes from adjacent crops and pastures.

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

Control

The best protection against aphid infestation and virus spread in chickpea is to prevent aphids from landing in the crop. This means controlling other sources, but also crop management and sowing into plant or stubble cover to reduce bare ground. Control the aphid population and its hosts beforehand. A prophylactic insecticide spray to prevent aphid incursion is not desirable.

Monitor chickpea and surrounding crops and pastures from early winter, and spray if plants with cowpea aphid can be found easily. Several insecticides for aphid control are highly toxic to bees and should not be applied while bees are foraging. [19]

7.18 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 green peach aphid, pea aphid, cowpea aphid and blue-green aphid, which will colonise pulse crops (Table 5). 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.

Green peach aphid and pea aphid are also important in spreading persistently transmitted viruses, depending on the virus involved.

Table 5: Differences in transmission of one persistent and two non-persistent viruses by four aphid species

CMV and BWYV are significant virus diseases in chickpea

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
Blue-green aphid	Yes	-	-

Source: GRDC Fact sheet 2010.

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

7.18.1 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 Pea seed-borne mosaic virus (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. Yarrum 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.

More information

https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2008/05/Principles-and-Implementation-of-IPM-in-Broadacre-Field-Crops-Risks-and-Benefits

https://grdc.com.au/Resources/Bookshop/2009/12/Integrated-Pest-Management-Fact-Sheet-National

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 lupins and chickpea.

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 green peach aphid, an important vector of BWYV, as most populations of green peach aphid are resistant. Monitor crops and neighbouring areas regularly. Identify the species of aphid present and their numbers.

Control the aphids if virus spread and direct feeding damage is of concern.

7.18.2 Controlling direct feeding damage

Monitoring crops for direct feeding damage can be worthwhile in most pulse crops, but less so in chickpea. Limited threshold information on aphid numbers is available for determining whether it is economically worthwhile to apply insecticides to prevent damage caused by feeding. Research in Western Australia suggests spraying if >30% of lupin growing tips are colonised by aphids from the flower bud stage through to podding, especially in aphid-susceptible varieties. Thresholds should be considered as a guide only, as many factors influence the economics of spraying.

Routine spraying of synthetic pyrethroid insecticides should be avoided because repeated applications of these insecticides can result in resistance developing in other non-target species (e.g. other aphids, mites) and will kill many natural insect predators.

A 'soft' insecticide is an option for controlling direct feeding damage when aphid populations are increasing.

It is not always possible to maintain a crop relatively clean of aphids. Allowing insects to build up to a sufficient threshold in an IPM approach may be acceptable for physical

More information

www.pulseaus.com.au

crop damage to foliage or even for grain quality. However, it may allow too great a spread of viruses in that crop, particularly early in the season. Most aphid build-up occurs in spring, which can also cause a problem with virus spread.

Some insects (e.g. cowpea aphid) are vectors for crop viruses (e.g. CMV). It takes only a few insect vectors, often undetectable, to cause a major virus problem in pulses, in particular BWYV in chickpea. Aphids do not colonise chickpea, but may spread viruses widely without being detected. The impact is seen well after any aphids have gone.

Insect vector control is only a component of a virus management strategy in pulses. Virus management aims at prevention that involves controlling the virus source, aphid populations and virus transmission into pulse crops. Elimination of summer weeds and self-sown pulses that act as a green bridge is important, as these host viruses and provide a refuge for aphid multiplication (Figure 23). Elimination of virus sources is critical (seed infection or other host species). Crop management is also important. Time of sowing, stubble cover, row spacing, density and crop health all play a role.

A prophylactic approach of spraying insecticides as a regular protectant is not necessarily the answer.[20]

Figure 23: Aphids colonised on a milk thistle plant in the middle of chickpea. This acts as a reservoir for aphid movement onto surrounding chickpea plants and virus infection. (Photo: G. Cumming, Pulse Australia)

More information

https://grdc.com.au/uploads/documents/Plague_Locusts_Factsheets.pdf

7.19 Australian plague locust *(Chortoicetes terminifera)*

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. Crops such as lupin that have epigeal emergence cannot recover if their cotyledons are eaten.

Most locust plagues originate in south-west Queensland and adjacent areas of South Australia, New South Wales and the Northern Territory. Locust populations develop following rainfall in this area.

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

More information

Victorian Department of Environment and Primary Industries: http://www.depi.vic.gov.au/agriculture-and-food/pests-diseases-and-weeds/pest-insects-and-mites/plague-locusts

NSW Department of Primary Industries: http://www.dpi.nsw.gov.au/agriculture/pests-weeds/insects/locusts

Department of Agriculture and Food Western Australia: https://www.agric.wa.gov.au/invasive-species/australian-plague-locust-overview

Australian Plague Locust Commission

With suitable conditions, autumn swarms may migrate 200–500 km into pastoral and adjacent agricultural areas. On arrival, they lay eggs, which produce the spring outbreak. [21]

Description

Adults of the Australian plague locust can be identified by their characteristic black spot on the tip of the hind wing (Figure 24). Nymphs or hoppers are more difficult to identify.

If they are in a large band, then it is likely to be the plague locust.

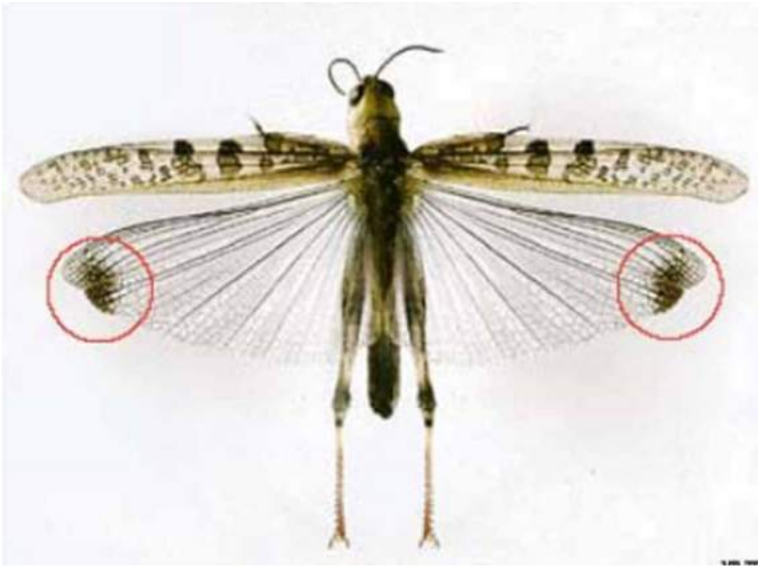

Figure 24: Australian plague locust. Note the black spot at the tip of the hind wing. (Photo: APLC via PIRSA)

Lifecycle

Adults are sexually mature within 2 weeks of developing wings. Females select suitable laying sites in the barest ground available e.g. roadsides, tracks.

Eggs are laid in pods at a depth of 20–50 mm. Each pod contains 30–50 pale-yellow eggs shaped like a banana, 5–6 mm long. Females lay up to four pods each before dying.

Eggs develop according to temperature and moisture. Eggs laid in autumn are usually dormant over winter and hatch in spring with soil temperature increases. Eggs laid in summer under ideal conditions may hatch within 14–16 days.

After hatching, nymphs or hoppers grow through five growth stages (Figures 25 and 26). Wing buds become progressively more notable through each stage.

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

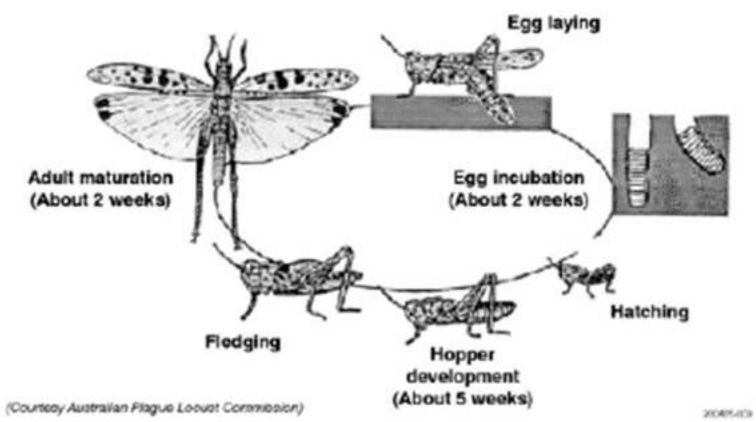

Figure 25: Life cycle of the Australian plague locust.

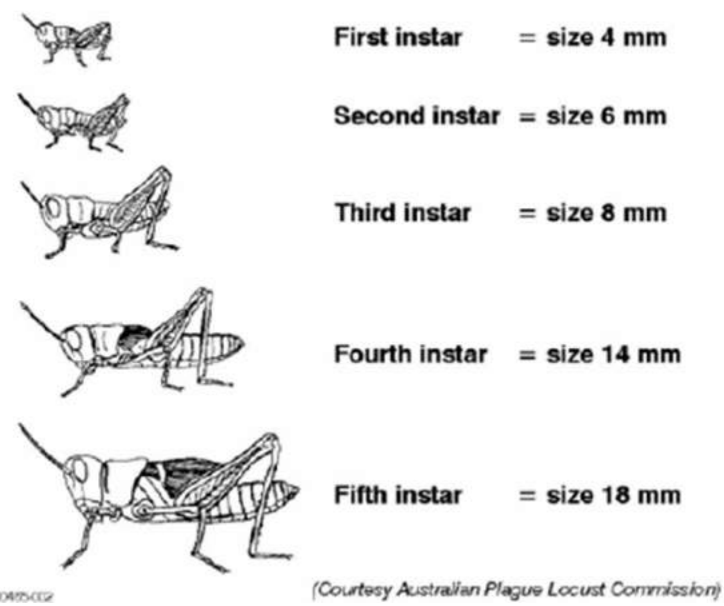

Figure 26: The five growth stages of plague locust nymphs.

Nymphs move away from egg beds and tend to concentrate into dense marching bands of size from a few square metres to several hectares. Bands may merge to increase to several kilometres with a distinct front. Older hoppers can travel up to 500 m in a single day. Hoppers complete their development in 4–6 weeks.

After their final moult, young adults emerge with fully developed wings. Milling flights increase over the band until the majority of hoppers have fledged. Adults concentrate into groups called swarms, which make low drifting flights up to 50 m high, and they can cover 10–20 km per day. Flight behaviour depends on the age of the adult, wind speed and temperature. Long-distance migration will occur at night if green feed has been available to enable fat accumulation.

Damage

Crops can be physically damaged, particularly seedlings. Rejection at grain delivery can occur if adult locusts, or parts of them, are present in the sample, or if objectionable stains and odour exist.

Control

The Australian Plague Locust Commission (APLC) undertakes surveillance threat assessments, forecasting and control measures when locust populations in outbreak areas have the potential to cross into agricultural areas.

In the event of a plague, local government may undertake some spraying operations (e.g. roadsides) within their own area. Where significant problems are expected government agencies may undertake large-scale control in pastoral and adjacent agricultural areas.

Effective suppression of locusts can only be achieved by cooperation between landowners, local governments and government agencies, combined with ongoing APLC activities.

Cultivation of egg beds will destroy the eggs. Use approved insecticides to target the bands of nymphs before they take flight. Advice on timings and chemicals can be obtained from state government departments or local chemical resellers. Often APVMA permits are required for chemical use. [22]

More information

www.apvma.gov.au

7.20 Spur-throated locust *(Austracris guttulosa)*

Spur-throated locust (Figures 27 and 28) is a pest of pastures, crops and some tree species. It is a tropical species of northern Australia, but extends its habitat into areas experiencing wet summer seasons.

It is often noticed in northern New South Wales, and in northern grain areas in Western Australia, but it rarely reaches damaging numbers. It is a declared pest insect in New South Wales.

More information

Comprehensive details of life cycle and controls are found at: http://www.dpi.nsw.gov.au/agriculture/pests-weeds/insects/locusts

https://www.agric.wa.gov.au/pest-insects/spur-throated-locust

QDAF (2015), Spur-throated locust

Figure 27: Adult-spur throated locust. (Photo: Australian Plague Locust Commission)

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

Figure 28: Adult spur-throated locust in a near mature lupin crop—note the pods. (Photo: K. Roberts NSW DPI)

Damage

Spur-throated locusts can cause damage to crops when they migrate in from neighbouring pastures and vegetation.

Life cycle

After hatching, nymphs or hoppers grow through six growth stages. Nymphs take 10 weeks to reach maturity. Nymphs do not form into large bands, so cannot be identified in the air, unlike the Australian plague locust.

Control

Control measures may be economic only in high-value crops or with high densities. Nymphs do not band and are generally quite scattered. Effective control will only be achieved if nymphs are also controlled adjoining pastures and vegetation to prevent re-invasion.

Ensure that product registration or a current APVMA permit exists before using any insecticide. [23]

More information

www.apvma.gov.au

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

7.21 Exotic chickpea insects

7.21.1 Exotic leaf miners (Diptera: Agromyzidae)

The Agromyzidae are a well-known group of small, similar flies whose larvae feed internally on living plant tissue, often as leaf and stem miners. Nearly all agromyzids are host-specific, but a few highly polyphagous species have become important pests of agriculture and horticulture in many parts of the world.

Key exotic agromyzid species for chickpeas include: chickpea leaf miner (*Liriomyza cicerina*) (Figure 29); American serpentine leaf miner (*Liriomyza trifolii*); and pea leaf miner (*Chromatomyia horticola*). The host range of these exotic species extends beyond chickpeas.

Description

Diagnostic characters are dealt with at the family level due to the extreme difficulty in differentiating species.

Adults are up to 2 mm in length, black/grey and yellow in colour with a conspicuous, bright yellow marking on the base of the thorax (scutellum) in most species. They are not very active flyers and tend to remain close to their target hosts. Adults lay eggs below the leaf surface.

Larvae are legless maggots up to 3 mm in length. They can be cream to yellowish in colour and are typically cylindrical in shape, tapering at the head region. There are three larval stages, which feed internally on plant tissue creating a tunnel or 'mine'. They can occasionally feed on the outside of pods.

Pupae look like tiny brown rice grains. *Liriomyza* species leave the plant to pupate in crop debris, soil or sometimes on the leaf surface, whereas *C. horticola* pupates inside the leaf at the end of the larval tunnel.

Damage

Larvae feed beneath the leaf surfaces, creating a winding tunnel or mine (Figures 30 and 31). Most leaf mines are greenish in colour at first, turning whitish over time. Some can also have distinct frass (waste) trails deposited in dark stripes on the sides of the mine (e.g. *L. trifolii*). Leaf mines wind irregularly through the leaf, increasing in width as larvae mature. Mine shapes in leaf tissue vary depending on species.

Leaf-miner damage is usually more severe in spring, with two to three generations occurring during the growing season.

Leaf-miner damage includes leaf destruction and retarded plant growth, and in severe infestations, total crop losses can occur, both from larval-mining and from leaf-puncturing caused by females ovipositing and sap-feeding. Infested plants are also susceptible to secondary attack by pathogenic fungi entering leaf punctures and mechanical transmission of plant viruses.

Surveillance

Leaf-mining is usually the first and most obvious symptom of the presence of leaf miners that can be seen in the field. Leaf-mining damage can also be caused by moth larvae, and exotic Agromyzidae species can be confused with native leaf miners. Any suspect mining should be sent for identification.

Entry Potential: Medium. Entry as eggs or larvae via imported plant material.

Establishment and Spread Potential: High.

Economic Impact: Medium.

Overall risk: Medium.

Initial incursions are likely to arise from horticultural areas and grains industry will face secondary attack. *Liriomyza* readily establish after introduction and rapidly spread.

Control is difficult. Economic impacts could be highly significant in most crops and across most cropping regions if eradication is not achieved. [24]

Figure 29: Chickpea leaf miner (Liriomyza cicerina). (Photo: A. Ames DPI Victoria, www.PaDIL.gov.au)

Figure 30: Larval-mining damage. (Photo: ' I Spy' Resource Manual (PaDIL))

[24] P Ridland, M Malipatil (2008) Industry Biosecurity Plan for the Grains Industry Threat Specific Contingency Plan. American serpentine leafminer, *Liriomyza trifolii*, bundled with *L. cicerina*, *L. huidobrensis*, *L. sativae*, *L. bryoniae* and *Chromatomyia horticola*. Plant Health Australia September 2008.

Figure 31: Larval-mining damage on daisy leaf caused by American serpentine leaf miner (L. trifolii). (Photo: Central Science Laboratory, Harpenden Archive, UK, bugwood.org)

7.22 Beneficial organisms

Beneficial organisms are important in overall pest management. Their impact in chickpea may be less than in other pulses because the malic acid in chickpea plants acts as a deterrent to many pest and beneficial insect species.

All pest populations are regulated to some degree by the direct effects of other living organisms. Beneficial organisms include a range of wasps, flies, bugs, mites, lacewings, beetles and spiders that can reduce insect pest populations through predation and parasitisation. Virus and fungal diseases also provide control.

A wide range of beneficial organisms can be grouped into three categories:

1. *Parasites*—organisms that feed on or in the body of another host. Most eventually kill their host and are free-living as an adult (parasitoids) (e.g. aphid wasp parasites).
2. *Predators*—mainly free-living insects that consume a large number of prey during their lifetime (e.g. shield bugs, lacewings, hover flies, spiders, predatory mites and predatory beetles).
3. *Insect diseases*—include bacterial, fungal and viral infections of insects.

Inappropriate use of an insecticide that reduces the number of beneficials can result in a more rapid build-up of insect populations and reliance on further use of insecticide. IPM in its simplest form is a management strategy in which a variety of biological, chemical and cultural control practices are combined to provide stable, long-term pest control.

A key aim of any IPM program is to maximise the number of beneficial invertebrates and incorporate management strategies other than pesticides that will help to keep pest insect numbers below an economic threshold.

Correct identification and regular monitoring are the cornerstone of IPM. When monitoring crops for insects, it is important to also check for the presence of, and record the build-up or decline in, the numbers of these beneficials, to make the best insect control decisions.

Integrate other pest management practices, together with the use of insecticides only where necessary, to maximise the number of beneficial organisms. This will result in better control of insect pest populations and a reduced reliance on the use of insecticide. [25]

A list of some beneficial organisms is provided below. For more details and photographs of beneficial organisms in insect management see:

CESAR web site: www.cesaraustralia.com/sustainable-agriculture/identify-an-insect/insect-gallery/

The GRDC 'Ute Guides' for specific pulse crops (lentil, field pea, faba bean and vetch) or insects

'I SPY: Insects of southern Australian broadacre farming systems identification manual and information resource' (Bellati *et al.* 2012)

Beetles
Carabid beetle (*Notonomus gravis*)
Transverse ladybird
Common ladybird

Bugs
Damsel bug (Nabidae)
Assassin bug
Glossy shield bug
Spined predatory shield bug (*Oechalia schellenbergii*)

Flies
Hoverfly (Syrphidae)
Tachinid fly

Lacewings
Green lacewing (Chrysopidae)
Brown lacewing (Hemerobiidae)

Mites
Pasture snout mite (*Bdellodes lapidaria*)
French anystis mite

Caterpillar wasps
Orange caterpillar parasite wasp
Two toned caterpillar wasp
Banded caterpillar wasp
Telenomus wasp
Orchid dupe
Trichogramma wasp
Braconid wasp (*Microplitis demolitor*)

Aphid wasps
Aphidius ervi
Trioxys complanatus wasp

Spiders
Wolf spider (Lycosidae)
Jumping spider (Salticidae)

Insect diseases—viral and fungal
Bacillus thuringiensis (BT)
Nuclear polyhedrosis virus (NPV)

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

Appendix 1: Predicted Helicoverpa development times in temperate regions

APPENDIX 1. Predicted heliothis development times based on HEAPS model simulations using long term average daily temperatures. Note that insects are presumed to begin at half-way through the initial stage on the starting date. Also note that in any given period temperatures may vary substantially from the long term averages, and therefore this table should be used as a guide only.

Starting date and stage		Central NSW Predicted days until...			North West NSW Predicted days until...			South West Qld Predicted days until...			Darling Downs Qld Predicted days until...			Central Highland Qld Predicted days until...		
		Large	Pupa	Moth	Large	Pupa	Moth	Large	Pupa	Moth	Large	Pupa	Moth	Large	Pupa	Moth
9 Sept	W. egg	34	53	78	34	52	74	29	45	64	32	50	70	22	35	51
	Small	13	38	64	12	35	62	12	30	52	12	34	59	9	24	42
	Medium	5	28	56	5	27	55	5	24	48	5	26	54	4	19	38
	Large	-	14	46	-	13	41	-	13	40	-	13	41	-	10	30
16 Sept	W. egg	32	50	75	32	49	69	27	42	60	30	47	66	20	31	47
	Small	12	34	61	12	34	59	10	28	48	12	32	56	9	22	39
	Medium	5	27	54	5	25	53	5	21	44	5	25	51	4	17	35
	Large	-	13	43	-	13	41	-	11	37	-	11	39	-	9	28
23 Sept	W. egg	30	47	70	30	47	66	24	38	56	27	44	63	19	31	46
	Small	12	33	59	11	32	56	9	26	46	11	29	51	7	20	36
	Medium	5	25	52	5	25	51	4	20	41	5	23	47	3	15	32
	Large	-	13	41	-	11	39	-	10	34	-	11	39	-	8	26
30 Sept	W. egg	29	47	64	28	43	62	22	35	52	27	42	61	18	29	43
	Small	10	30	55	12	31	53	9	25	43	9	27	47	7	18	34
	Medium	5	24	51	5	24	48	4	19	39	5	21	44	3	15	31
	Large	-	10	38	-	10	38	-	9	31	-	10	36	-	7	24
7 Oct	W. egg	27	44	63	26	42	60	21	33	49	25	39	58	16	27	41
	Small	11	29	54	10	27	48	9	23	41	9	26	45	6	18	33
	Medium	5	23	48	4	22	44	4	18	36	4	20	40	3	14	29
	Large	-	11	38	-	10	36	-	9	29	-	10	34	-	7	23
14 Oct	W. egg	26	41	59	24	39	57	19	31	46	23	37	56	16	26	40
	Small	9	26	49	10	27	46	8	21	38	9	25	44	6	17	31
	Medium	4	20	44	4	20	41	3	16	34	4	19	38	3	14	28
	Large	-	10	37	-	10	34	-	8	27	-	9	31	-	7	22
21 Oct	W. egg	24	39	55	23	36	53	19	30	44	20	33	52	15	25	39
	Small	9	27	46	9	25	43	7	19	35	9	23	42	6	16	30
	Medium	4	19	42	4	19	38	3	15	32	4	18	37	3	13	27
	Large	-	9	34	-	9	31	-	8	26	-	9	28	-	6	21
28 Oct	W. egg	23	37	54	20	33	49	17	28	42	19	32	49	15	25	38
	Small	9	25	44	8	23	41	6	18	33	7	21	40	6	16	30
	Medium	4	20	39	3	18	36	3	14	30	4	16	35	3	12	26
	Large	-	9	34	-	7	28	-	7	24	-	8	27	-	6	20
4 Nov	W. egg	22	35	50	19	31	46	17	27	41	19	31	47	15	25	37
	Small	9	25	41	8	21	38	7	18	32	7	20	39	6	16	29
	Medium	3	18	37	3	17	35	3	14	28	3	16	35	2	12	26
	Large	-	10	30	-	8	27	-	7	23	-	8	27	-	6	20
11 Nov	W. egg	21	33	49	18	30	44	15	25	38	19	31	45	15	24	36
	Small	9	23	40	7	20	36	6	17	31	7	20	37	6	16	28
	Medium	3	18	34	3	15	32	3	13	27	3	15	33	2	12	25
	Large	-	9	28	-	8	26	-	7	21	-	8	27	-	6	20

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SECTION 8

Nematode management

More information

[K Moore, K Hobson, S Harden, L Jenkins, R Brill (2014), Chickpea yields with and without *Pratylenchus thornei* – Coonamble & Trangie 2013 p128]

[B Burton, L Bailey, K Adhikari (2016), Impact from Pratylenchus thornei, Macalister 2015]

Root-lesion nematodes (RLN; *Pratylenchus* spp.) are microscopic, worm-like animals that extract nutrients from plants, causing yield loss. In the northern grains region, the predominant RLN, *P. thornei*, costs the wheat industry AU$38 million[1] annually, and including the secondary species, *P. neglectus*, RLN are found in three-quarters of fields tested.

Intolerant crops such as wheat and chickpeas can lose 20–60%[2] in yield when nematode populations are high. Resistance and susceptibility of crops can differ for each RLN species; for example, sorghum is resistant to *P. thornei* but susceptible to *P. neglectus*. A tolerant crop yields well when large populations of RLN are present (the opposite is intolerance). A resistant crop does not allow RLN to reproduce and increase in number (the opposite is susceptibility). [3]

Successful management relies on:

- farm hygiene to keep fields free of RLN
- growing tolerant varieties when RLN are present, to maximise yields
- rotating with resistant crops to keep RLN at low levels[4]

8.1 Background

More information

https://grdc.com.au/Resources/Factsheets/2015/03/Root-Lesion-Nematodes

Root-lesion nematodes use a syringe-like 'stylet' to extract nutrients from the roots of plants (Figure 1). Plant roots are damaged as RLN feed and reproduce inside plant roots. *Pratylenchus thornei* and *P. neglectus* are the most common RLN species in Australia. In the northern grains region, *P. thornei* is the predominant species but *P. neglectus* is also present. These nematodes can be found deep in the soil profile (to 90 cm depth) and are found in a broad range of soil types, from heavy clays to sandy soils. Wheat is susceptible to both *P. thornei and P. neglectus*. [5]

New CSIRO research funded by the Grains Research and Development Corporation (GRDC) is examining how nematodes inflict damage by penetrating the outer layer of wheat roots and restricting their ability to transport water.

1 GM Murray, JP Brennan (2009) The current and potential costs from diseases of wheat in Australia. Grains Research and Development Corporation Report. https://www.grdc.com.au/~/media/B4063ED6F63C4A968B3D7601E9E3FA38.pdf

2 KJ 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 Grains Research Update, July 2013.

3 KJ Owen, J Sheedy, N Seymour (2013) Root lesion nematode in Queensland. Soil Quality Pty Ltd Fact Sheet.

4 R Daniel (2013) Managing root-lesion nematodes: how important are crop and variety choice? Northern Grower Alliance/GRDC Update Paper, 16/07/2013.

5 KJ Owen, J Sheedy, N Seymour (2013) Root lesion nematode in Queensland. Soil Quality Pty Ltd Fact Sheet.

Fact sheets

[Soil Quality Pty Ltd nematode survey results]

[GRDC Parasitic Plant Nematodes (Northern Region Fact Sheet)]

http://www.daf.qld.gov.au/__data/assets/pdf_file/0010/58870/Root-Lesion-Nematode-Brochure.pdf

Figure 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: Sean Kelly, Department of Agriculture and Food, Western Australia)

8.2 Symptoms and detection

Root-lesion nematodes 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 RLN is to have soil tested in a laboratory. Fee-for-service testing of soil offered by the PreDicta B root disease testing service of the South Australian Research and Development Institute (SARDI) can determine levels of *P. thornei* and *P. neglectus* present. [6]

Similar results can be obtained by soil testing either by manual counting (under microscopes) or by DNA analysis (PreDicta B), with commercial sampling generally at depths of 0–15 or 0–30 cm. [7]

P. thornei populations greater than 40,000 per kg at harvest will require a double break of around 40 months free of a host to reduce the population below the accepted threshold of 2000 *Pt*/kg.

P. thornei populations greater than 10,000 per kg at harvest will require a single break of around 30 months free of a host to reduce the population below the accepted threshold of 2000 *Pt*/kg. [8]

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 at 0–15 or 0–30 cm depth.

6 KJ Owen, J Sheedy, N Seymour (2013) Root lesion nematode in Queensland. Soil Quality Pty Ltd Fact Sheet.

7 R Daniel (2013) Managing root-lesion nematodes: how important are crop and variety choice? Northern Grower Alliance/GRDC Update Paper, 16/07/2013.

8 J Whish, J Thompson (2016), How long does it take to reduce Pratylenchus thornei (Root lesion nematode) population in the soil? https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/02/How-long-does-it-take-to-reduce-Pratylenchus-thornei-populations-in-the-soil

Signs of nematode infection in roots include dark lesions or poor root structure. The damaged roots are inefficient at taking up water and nutrients—particularly nitrogen (N), phosphorus (P) and zinc (Zn)—causing symptoms of nutrient deficiency and wilting in the plant shoots. Intolerant wheat varieties may appear stunted, with yellowing of lower leaves and poor tillering (Figure 2). These symptoms may not be present in other susceptible crops such as barley and chickpea. [9]

Figure 2: Symptoms of root-lesion nematode infection of an intolerant wheat variety include yellowing of lower leaves, decreased tillers and wilting. There are no obvious symptoms in the susceptible chickpea and faba bean plots on either side of the wheat. (Photo: Kirsty Owen, DAFF)

Testing service

http://www.sardi.sa.gov.au/products__and__services/entomology/diagnostic_services/predicta_b

8.3 Symptoms in chickpeas

Severely affected plants are stunted and may have some yellowing of their foliage, but often have no obvious foliar symptoms of disease. Diseased plants usually have shorter lateral roots and fewer root hairs.

Where many nematodes invade chickpea roots, the affected tissues sometimes turn dark brown–black, giving alternating sections of healthy and discoloured root tissue. Often discoloration may appear as brown or black stripes along the roots. In severe cases young roots die.

Microscopic examination of the root system is required to confirm the presence of the nematode (Figure 3).

Pratylenchus impair root function, limiting water and nutrient uptake by the plant. Affected plants may show general unthriftiness, or symptoms of nitrogen deficiency. Symptoms are increased when plants are subjected to water and nutrient stress, or if there is also root damage caused by fungi.

Symptoms of infection on root systems include:

- disintegration of outer layers of root tissue
- reduction in root hairs and/or nodules
- a lack of or stunting of side (lateral) roots
- brown lesions and discoloration of roots

9 KJ Owen, J Sheedy, N Seymour (2013) Root lesion nematode in Queensland. Soil Quality Pty Ltd Fact Sheet.

Root symptoms are often difficult to diagnose in the field and are usually not seen until plants are older than 8 weeks. Root symptoms are generally more obvious in plants grown in sandier soils. [10]

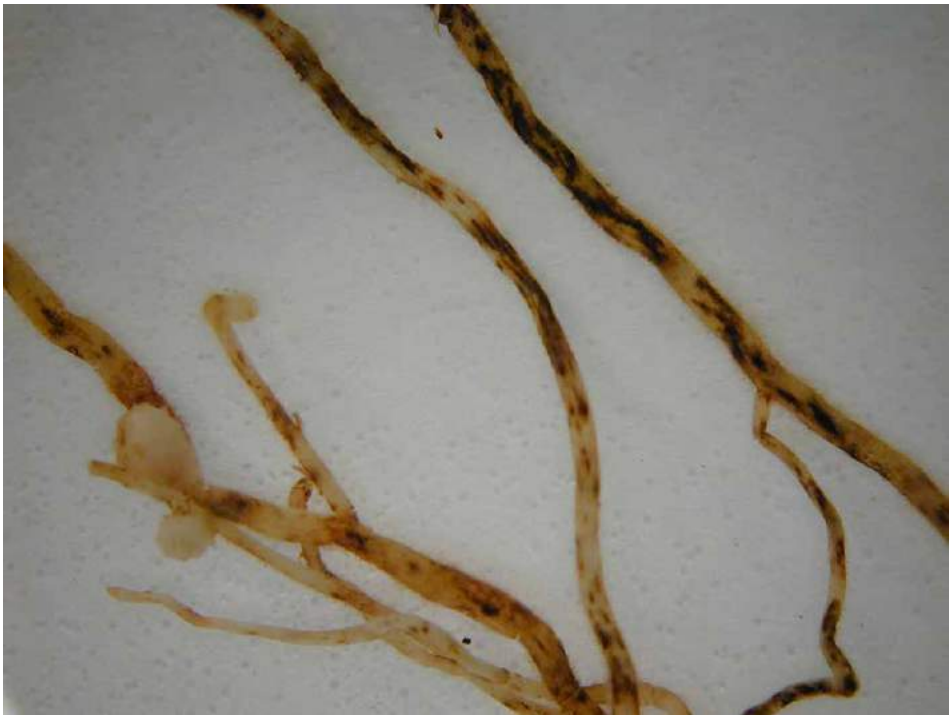

Figure 3: Pratylenchus (root-lesion nematode) lesions on chickpea roots. (Photo: SARDI)

8.3.1 What is seen in the paddock?

Although symptoms of RLN damage in wheat can be dramatic, they can easily be confused with nutritional deficiencies and/or moisture stress.

Damage from RLN is in the form of brown root lesions but these can be difficult to see or can also be caused by other organisms. Root systems are often compromised, with reduced branching, reduced quantities of root hairs and an inability to penetrate deeply into the soil profile. The RLN create an inefficient root system that reduces the ability of the plant to access nutrition and soil water.

Visual damage above ground from RLN is non-specific. Yellowing of lower leaves is often observed, together with reduced tillering and a reduction in crop biomass. Symptoms are more likely to be observed later in the season, particularly when the crop is reliant on moisture stored in the subsoil.

In the early stages of RLN infection, localised patches of poorly performing wheat may be observed. Soil testing of these patches may help to confirm or eliminate RLN as a possible issue. In paddocks where previous wheat production has been more uniform, a random soil-coring approach may be more suitable. Another useful indicator of RLN presence is low yield performance of RLN-intolerant wheat varieties. [11]

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

[11] R Daniel (2013) Managing root-lesion nematodes: how important are crop and variety choice? Northern Grower Alliance/GRDC Update Paper, 16/07/2013.

8.4 Management

There are four key strategies for the management of RLN (Figure 4):

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 wheat varieties to maximise yields (www.nvtonline.com.au). Tolerant varieties grow and yield well when RLN are present.
4. Rotate with resistant crops to prevent increases in RLN (Table 1, Figure 5). 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. [12]

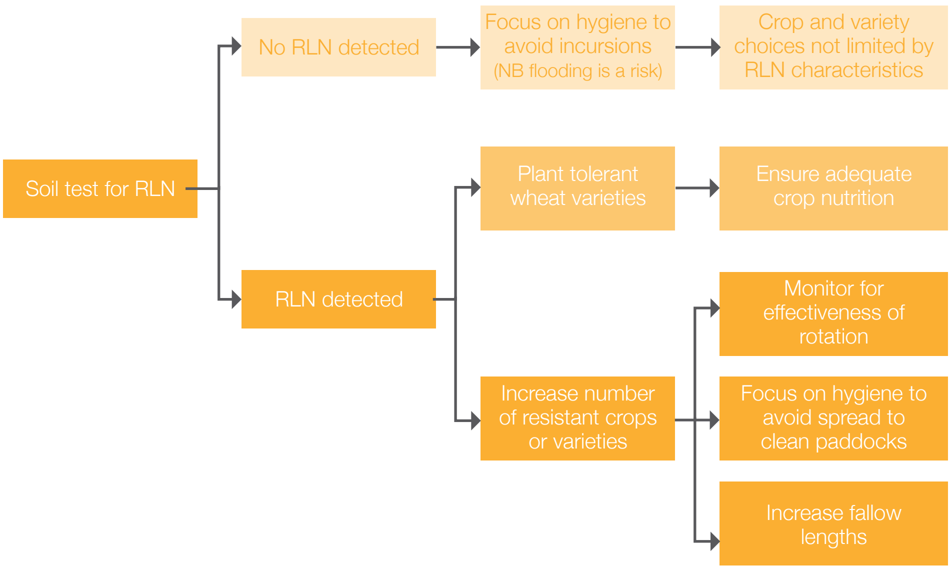

Figure 4: Root-lesion nematode management flow-chart

Other considerations include:

- **Nematicides.** There are no registered nematicides for RLN in broadacre cropping in Australia. Screening of potential candidates is conducted, but RLN are a very difficult target, with populations frequently deep in the soil profile.
- **Nutrition.** Damage from RLN reduces the ability of cereal roots to access nutrients and soil moisture and can induce nutrient deficiencies. Under-fertilising is likely to exacerbate RLN yield impacts; however, over-fertilising is unlikely to compensate for a poor variety choice.
- **Variety choice and crop rotation.** These 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 on RLN build-up). Note that varieties and crops often have varied tolerance and resistance levels to *P. thornei* and *P. neglectus*.
- **Fallow.** Populations of RLN will decrease during a 'clean' fallow, but the process is slow and expensive in lost 'potential' income. Additionally, long fallows may decrease arbuscular mycorrhiza (AM) levels and create more cropping problems than they solve. [13]

More information

J Whish, J Thompson (2016), How long does it take to reduce Pratylenchus thornei (Root lesion nematode) population in the soil?

N Seymour, G Stirling, J Li (2016), Biological suppression of root lesion nematodes in northern grain growing soils

T Dixon, E Berry (2015), Can wild chickpeas reduce the nematode problem in Australian crops?

[12] KJ Owen, J Sheedy, N Seymour (2013) Root lesion nematode in Queensland. Soil Quality Pty Ltd Fact Sheet.

[13] R Daniel (2013) Managing root-lesion nematodes: how important are crop and variety choice? Northern Grower Alliance/GRDC Update Paper, 16/07/2013.

Table 1: Susceptibility and resistance of various crops to root-lesion nematodes[14]

RLN species	Susceptible	Intermediate	Resistant
P. thornei	Wheat, chickpea, faba bean, barley, mungbean, navy bean, soybean, cowpea	Canola, mustard, triticale, durum wheat, maize, sunflower	Canary seed, lablab, linseed, oats, sorghum, millet, cotton, pigeon pea
P. neglectus	Wheat, canola, chickpea, mustard, sorghum (grain), sorghum (forage)	Barley, oat, canary seed, durum wheat, maize, navy bean	Linseed, field pea, faba bean, triticale, mungbean, soybean

Figure 5: Crop rotation to manage root-lesion nematodes depends on the nematode species present in your field. Mungbeans (left) are susceptible to P. thornei but resistant to P. neglectus. By contrast, sorghum (right) is resistant to P. thornei but susceptible to P. neglectus. (Photo: Kirsty Owen, DAFF)

8.4.1 Crop Rotation

P. neglectus was found in 32% of paddocks (often in combination with *P. thornei*) in the northern region in a survey of 800 paddocks (Thompson *et al.* 2010). 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.

GRDC Update Paper

https://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/07/Summer-crop-decisions-and-root-lesion-nematodes

In a field experiment, populations of *P. neglectus* increased after growing grain sorghum. Populations increased from 3.1 times after MR32⏀ (4,400 *P. neglectus*/kg soil) to 7.3 times after MRGoldrush⏀ (10,400 *P. neglectus*/kg soil) compared to soil at planting (1,400 *P. neglectus*/kg soil). [15]

Summer crops 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. [16]

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[14] KJ Owen, J Sheedy, N Seymour (2013) Root lesion nematode in Queensland. Soil Quality Pty Ltd Fact Sheet.

[15] http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/07/Summer-crop-decisions-and-root-lesion-nematodes

[16] 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 Grains Research Update, July 2013.

8.4.2 Sowing time

Wheat variety choice can have a great impact on yield loss to *P. thornei* (up to 43% yield loss in intolerant bread wheat varieties in 2011), and yield losses from *P. thornei* can be exacerbated by delayed sowing and drier conditions. [17]

New South Wales Department of Primary Industries (NSW DPI) winter cereal time-of-sowing trials at Coonamble, Mungindi, Trangie, Come-by-Chance and Gurley, NSW, in 2011 showed the following:

- Winter crop type and variety choice have a large effect on the build-up of nematode populations in the soil due to differences in their resistance to *P. thornei*.
- This was most pronounced in bread wheat where the variety choice:
  - increased the *P. thornei* population by 1.8–3.6 times (9737 up to 19,719 *P. thornei*/kg soil) at Coonamble, and
  - decreased the *P. thornei* population by 64% between the most susceptible and most resistant varieties at Mungindi (25,448 *v.* 9050 *P. thornei*/kg soil).
- *Pratylenchus thornei* populations were six times larger in the most susceptible variety, Lincoln⊕, than in the most resistant variety, Gauntlet⊕, at Trangie.
- Earlier sowing generally increased the build-up of *P. thornei* populations at Trangie, especially in the most susceptible variety.
- The build-up of *P. thornei* populations in the field trial is broadly in line with published resistance ratings, but discrepancies appear to exist, especially with LongReach Spitfire⊕, which appears better than its current rating of very susceptible.
- Both *P. thornei* and crown rot (caused by *Fusarium pseudograminearum*) cause significant yield loss in intolerant/susceptible varieties alone or in combination, as shown at Gurley.
- *Pratylenchus thornei* and crown rot did not reduce grain protein levels at the Gurley site.
- Some recently released varieties appear to combine improved tolerance to *P. thornei* with increased resistance to crown rot, which provided a yield advantage of up to 109% at the Gurley site in 2012.
- Reliable resistance ratings appear to be produced under both large and moderate starting populations of *P. thornei* at Mungindi. Hence, National Variety Trials (NVT) are a potentially useful source of reliable field-based assessments. [18] Visit www.nvtonline.com.au

More information

Northern Grains Region trial results autumn 2013

L Serafin, S Simpfendorfer, M Sissons, A Verrell, G McMullen (2015), Northern grains region trial results Autumn 2015

Delayed sowing

In two trials conducted in 2011, *P. thornei* was demonstrated to reduce yield by up to 43% under large starting populations with delayed sowing and drier growing conditions. Delayed sowing into late autumn/winter is likely to see crops initially develop under cooler soil temperatures, thus reducing the rate of root development. Conversely, earlier sown crops establish under warmer soil conditions and have more rapid, early root growth if adequate moisture is available.

Drier soil conditions during crop establishment and early growth, for example with the second sowing time (22 June) at Coonamble in 2011, are also likely to restrict early root development. In theory, any restriction to root development is likely to inhibit a crop's ability to compensate for *P. thornei* feeding upon these root systems. Variety choice can have a large impact on yield and, hence, profitability when cropping in soils with large populations of *P. thornei*. To date, these trials have only examined the relative tolerance of varieties to *P. thornei*. It should be stressed that a variety's resistance to *P.*

17 S Simpfendorfer, M Gardner, G McMullen (2012) Impact of sowing time and varietal tolerance on yield loss to the root-lesion nematode *Pratylenchus thornei*. GRDC Grains Research Update, Goondiwindi, March 2012.

18 NSW DPI (2013) Northern Grains Region trial results autumn 2013. NSW Department of Primary Industries.

thornei (build-up of nematode populations within the soil) should also be an important consideration in variety choice. [19]

Interaction with crown rot

Crown rot remains a significant disease in the region, with losses dependent on soil moisture and temperature stress experienced during flowering and grain-fill. Crown rot caused yield losses of up to 37% in durum varieties at the Coonamble site in 2011, but cooler, wetter conditions limited the expression (yield loss) of this disease at Mungindi in 2011. Averaged across the different winter cereal types, crown rot reduced yield by 18% in barley, 27% in durum wheat and 22% in bread wheat at Coonamble in 2011. Research conducted by NSW DPI and the Northern Grower Alliance (NGA) across 11 sites in northern NSW in 2007 demonstrated that crown rot caused average yield losses of 20% in barley (up to 69% under drier conditions and hotter temperatures during grain-fill), 25% in bread wheat (up to 65%) and 58% in durum (up to 90%).

The Coonamble site trial demonstrates that the tolerance of wheat varieties to crown rot does not appear to be related to their level of tolerance to *P. thornei*. Yield losses to both diseases in intolerant varieties can be significant (up to 43% for *P. thornei* and up to 37% for crown rot at Coonamble in 2011) under high levels of inoculum. However, the benefit obtained from sowing a more tolerant bread wheat variety appears greater for *P. thornei* (up to 43%) than for crown rot (up to 21%). Another way of expressing this is that the difference in tolerance levels between wheat varieties appears larger for *P. thornei* than for crown rot. [20]

GRDC Update Paper

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2012/04/Impact-of-sowing-time-and-varietal-tolerance-on-yield-loss-to-the-rootlesion-nematode-pratylenchus-thornei

Selecting tolerant varieties

Selecting tolerant wheat varieties is one of the main options for maintaining profit in the presence of high populations of *P. thornei*. By contrast, even the most crown rot resistant/tolerant commercial wheat variety can still suffer up to 50% yield loss under high levels of inoculum when hot/dry conditions occur during grain-fill. Variety selection is not a primary strategy for managing crown rot. Hence, where soil populations of *P. thornei* are large, more emphasis should be placed on a wheat variety's tolerance to *P. thornei* than to crown rot. Rotation to non-host crops remains the primary management tool for crown rot and can also be a valuable strategy to reduce or maintain *P. thornei* populations below the threshold (<2,000 *P. thornei*/kg soil) for yield loss in intolerant wheat varieties. [21]

Survey results

www.nga.org.au/results-and-publications/download/49/surveys/root-lesion-nematode-survey.pdf

Current industry knowledge

In 2010, the NGA conducted a survey of current levels of knowledge about nematodes (particularly RLN) in northern broadacre farming systems and the management practices being employed. The results are being used to prioritise research and development activity.

8.5 What are resistance and tolerance?

8.5.1 Resistance: nematode multiplication

- Resistant crops do not allow RLN to reproduce and increase in number in their roots.
- Susceptible crops allow RLN to reproduce so that their numbers increase. Moderately susceptible crops allow increases in nematode populations but at a slower rate.

8.5.2 Tolerance: crop response

- Tolerant varieties or crops yield well when sown in fields containing large populations of nematodes.

[19] NSW DPI (2013) Northern Grains Region trial results autumn 2013. NSW Department of Primary Industries.

[20] NSW DPI (2013) Northern Grains Region trial results autumn 2013. NSW Department of Primary Industries.

[21] S Simpfendorfer, M Gardner, G McMullen (2012) Impact of sowing time and varietal tolerance on yield loss to the root-lesion nematode *Pratylenchus thornei*. GRDC Grains Research Update, Goondiwindi, March 2012.

- Intolerant varieties or crops yield poorly when sown in fields containing large populations of nematodes.

Most pulses have good resistance to both species of *Pratylenchus* and so reduce nematode populations in cropping rotations; exceptions are chickpeas (with both species), and vetch (with *P. thornei*) (Table 2). [22]

Table 2: Resistance and tolerance ratings of pulses to root-lesion nematode

Chickpea varieties have a range of resistance and tolerance to Pratylenchus species. S, Susceptible; R, resistant; I, intolerant; T, tolerant; M, moderately; V, very

Pulse	*Pratylenchus neglectus*		*Pratylenchus thornei*	
	Resistance	Tolerance	Resistance	Tolerance
Chickpeas	S–MR	MI–T	VS–R	MI–T
Faba beans	R	–	MR	MI
Field peas	R	–	R	T
Lentils	R	T	R	MT
Vetch var. Blanchefleur	MR	T	S	I–MI
Vetch var. Languedoc	MR	T	MS	I–MI
Vetch var. Morava	MR	T	MS	I–MI

8.6 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) (Figure 6.)

There are four possible combinations of resistance and tolerance:	
Tolerant–resistant e.g. sorghum cv. MR43 to *P. thornei* *and wheat breeding lines released for development*	**Tolerant–susceptible** e.g. wheat cv. EGA Gregory to *P. thornei*
Intolerant–resistant *No commercial wheat lines in this category*	**Intolerant–susceptible** e.g. wheat cv. Strzelecki to *P. thornei*

Figure 6: Combinations and examples of tolerance and resistance[23]

Tolerance and resistance of wheat varieties to RLN are published each year at www.nvtonline.com.au or in *Wheat varieties for Queensland*.

Current GRDC-funded research by the NGA and NSW DPI is examining the importance of crop and variety choice. The NGA has run large and complex trials and results are outlined in the GRDC Update Paper.

Growers are advised to recognise that there are consistent varietal differences in *P. thornei* and *P. neglectus* resistance within wheat and chickpea varieties; to avoid crops or varieties that allow the build-up of large populations of RLN in infected paddocks; and to monitor the impact of rotations.

The DAFF and NSW DPI wheat variety guides detail the level of variety tolerance to both species of RLN. Selection of wheat varieties based on these published RLN tolerance rankings is critical to avoid significant yield losses, particularly in paddocks with large populations of *P. thornei*.

22 QPIF (2009) Root lesion nematodes. Management of root lesion nematodes in the northern grain region. Department of Agriculture, Foresty and Fisheries, http://www.daf.qld.gov.au/__data/assets/pdf_file/0010/58870/Root-Lesion-Nematode-Brochure.pdf

23 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 Grains Research Update, July 2013.

GRDC-funded researchers are currently incorporating *P. thornei* resistances found in a wheat line selected from the variety Gatcher⦻ and some wheat landraces from West Asia and North Africa into pre-breeding efforts. Excellent resistance to *P. thornei* and *P. neglectus* has been found in synthetic hexaploid wheats.

Resistances are being incorporated into some of the most tolerant wheat varieties, including EGA Gregory⦻ and EGA Wylie⦻, to produce parents that are adapted to the northern region. [24]

8.6.1 Tolerance

Wheat breeding has provided a number of varieties with moderate or higher levels of tolerance to *P. thornei*, e.g. Sunvale⦻, Baxter⦻, EGA Wylie⦻ and EGA Gregory⦻. These varieties will reduce the level of yield loss due to *P. thornei*.

At a trial site near Yallaroi in 2012, a range of crops and varieties was grown and performance evaluated under relatively 'low' and 'high' starting population densities of *P. thornei* (about 2,000 and 19,000 nematodes/kg soil). Figure 7 shows the impact of *P. thornei* on yield of varieties with a range of tolerance levels.

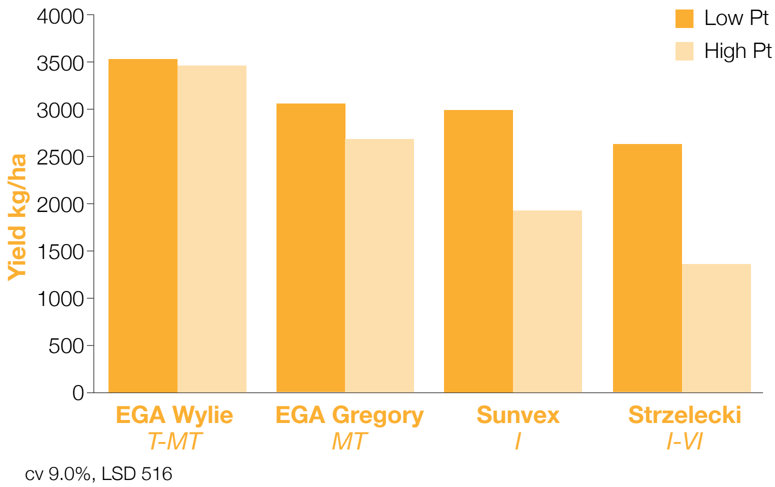

Figure 7: Comparison of wheat variety yields under 'low' and 'high' starting population densities of P. thornei (Pt) near Yallaroi 2012 (Trial RH1213)

*Indicates significant yield difference within a variety between 'low' and 'high' *P. thornei* strips at *P* = 0.05.

Codes below variety names are the DAFF published ratings of *P. thornei* tolerance: T, tolerant; MT, moderately tolerant; I, intolerant; VI, very intolerant.

NB: What was categorised as the 'low' starting population density of *P. thornei* was still equal to the current industry threshold. At this level, significant yield losses (up to 20%) may occur in intolerant wheat varieties. Consequently, the measured yield impact between 'low' and 'high' *P. thornei* in this trial is an underestimate of the full *P. thornei* affect. [25]

The varieties rated as *P. thornei* intolerant (Strzelecki⦻ and Sunvex⦻) suffered significant yield reductions of 35–48 % in this trial when grown in the 'high' *P. thornei* plots. Yield losses of about 1–1.25 t/ha were recorded, with economic losses >$250/ha. The two varieties that were more tolerant (EGA Wylie⦻ and EGA Gregory⦻) did not suffer a significant yield reduction.

Choosing tolerant varieties will limit the yield and economic impact from *P. thornei*; however, some of these varieties still allow high levels of nematode build-up. The second issue to be considered is variety resistance/susceptibility. [26]

[24] J Thompson, J Sheedy, N Robinson, R Reen, T Clewett, J Lin (2012) Pre-breeding wheat for resistance to root-lesion nematodes. GRDC Grains Research Update, Goondiwindi, March 2012.

[25] K Owen, J Sheedy, N Seymour (2013) Root lesion nematode in Queensland. Soil Quality Pty Ltd Fact Sheet.

[26] R Daniel (2013) Managing root-lesion nematodes: how important are crop and variety choice? Northern Grower Alliance/GRDC Update Paper, 16/07/2013.

8.6.2 Resistance

Figure 8 shows the mean *P. thornei* population remaining after a range of winter crops was grown in 2011. Although all crops were sown in individual trials (to enable weed and pest control), the data should generally reflect the resistance differences between these crops. [27]

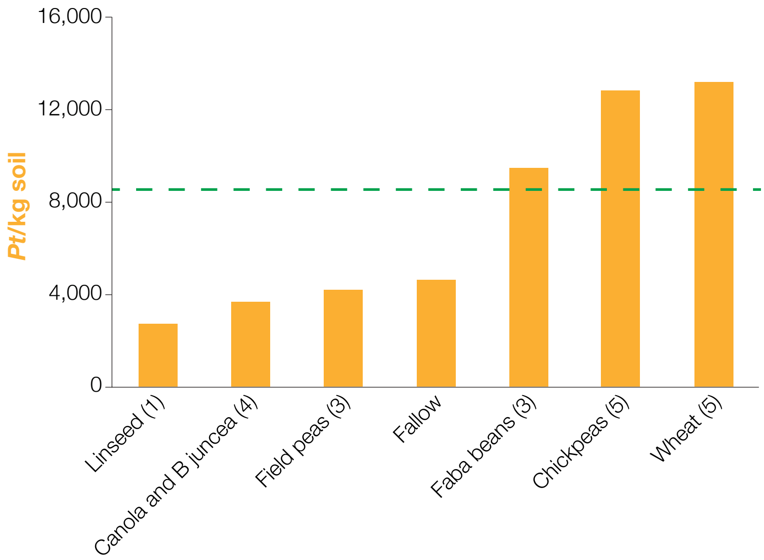

Figure 8: Comparison of P. thornei (Pt) population remaining in March–April 2012 following different winter crop species near Weemelah, NSW, 2011 (Trials RH1101–1109). Values in parentheses are the number of varieties of each crop. The solid blue horizontal line indicates the mean P. thornei level in March 2011.

More information

https://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/07/Managing-root-lesion-nematodes-how-important-are-crop-and-variety-choice

http://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Latest-nematode-summer-and-winter-crop-rotation-results

Sorghum is generally characterised as resistant to *P. thornei*. The NGA conducted sampling of eight sorghum variety trials in summer 2012–13. Across all sites, there was mean reduction of 45% in *P. thornei* population following a single sorghum crop. Sorghum is a very useful option to assist in *P. thornei* management.

Pratylenchus thornei does not die out completely even after five successive resistant crops. Management of RLN is ongoing and requires regular soil tests to monitor nematode populations.

8.6.3 Resistance differences between Desi chickpea varieties

Recent field data are showing consistent differences in Pt resistance between commercial chickpea varieties. Figure 9 shows a summary of the performance of a range of chickpea varieties in 9 trials, during 2010-2014, conducted by DAFF QLD, NSW DPI or NGA.

[27] R Daniel (2013) Managing root lesion nematode: how important are crop and variety choice? GRDC Update papers July 2013, https://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/07/Managing-root-lesion-nematodes-how-important-are-crop-and-variety-choice

More information

K Moore, S Harden, R Brill, N Coombes (2013) Increasing numbers of the root lesion nematode *Pratylenchus thornei* did not affect yield of six chickpea varieties— Coonamble 2012.

K Moore, K Hobson, S Harden, L Jenkins, R Brill (2013) Effect of the root lesion nematode *Pratylenchus thornei* on chickpea yield— Coonamble & Trangie 2012.

Both references at: http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0004/468328/Northern-grains-region-trial-results-autumn-2013.pdf

https://www.daf.qld.gov.au/plants/field-crops-and-pastures/broadacre-field-crops/crop-diseases/root-lesion-nematode

B Burton, Northern Grower Alliance (2015), Impact of crop varieties on RLN multiplication

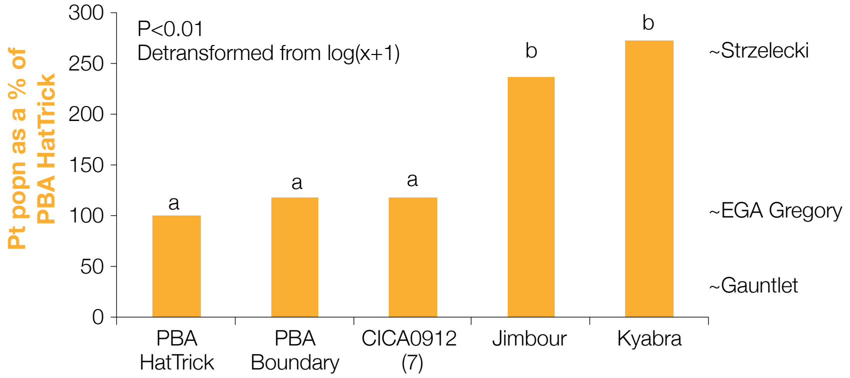

Figure 9: Comparison of Pt population remaining between Desi varieties as a % of PBA HatTrick, 2010-2014. All varieties evaluated in all 9 trials except CICA0912 (only 7 trials).

The position of the wheat varieties on the RHS of the graph indicate the best current estimate of comparison between these varieties for Pt build-up. This data has been generated where Desi chickpeas and wheat have been grown at the same trial site.

Key point: All Desi varieties evaluated to date appear to provide a medium to high risk of Pt build-up. Mean Pt populations after Jimbour and Kyabra have been generally double the level compared to the population remaining after growing either PBA HatTrick or PBA Boundary. Growers with Pt infestations should certainly avoid varieties that support higher populations of Pt.

8.6.4 Resistance differences between Kabuli chickpea varieties

Field data has not shown any consistent differences in resistance between the current commercial Kabuli varieties (Figure 10). In general, Kabuli varieties appear to be leaving behind a similar Pt population to the highly susceptible Desi chickpea varieties such as Jimbour and Kyabra. This would suggest all current commercial Kabuli varieties are in the high risk category for Pt build-up.

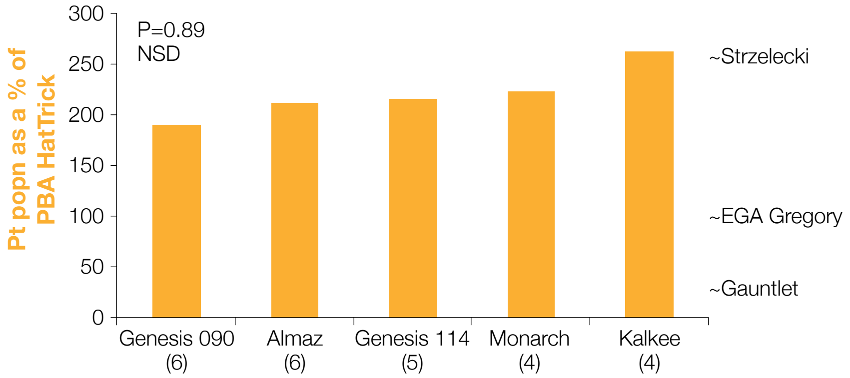

Figure 10: Comparison of Pt population remaining between Kabuli varieties as a % of PBA HatTrick, 2010-2014. (Number) indicates the number of field trials in which the variety was evaluated.

NSD = No significant difference between treatments. The position of the wheat varieties on the RHS of the graph indicate our best current estimate of comparison between these varieties for Pt build-up. This data has been generated where Kabuli chickpeas and wheat have been grown at the same trial site. [28]

[28] B Burton, Northern Grower Alliance (2015), Impact of crop varieties on RLN multiplication. https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2015/03/Impact-of-crop-varieties-on-RLN-multiplication

More information

http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0003/431265/Cereal-pathogen-survey.pdf

8.7 Damage caused by nematodes

Pratylenchus thornei is widespread in the northern grains region, with surveys conducted by DAFF and NSW DPI showing its presence in 50–70% of paddocks. It is frequently at concerning levels, being found at >2,000 individuals/kg soil in about 20 to 30% of paddocks.

Yield losses in wheat of up to 50% are not uncommon when *P. thornei*-intolerant wheat varieties are grown in paddocks infested with *P. thornei*. Yield losses in chickpeas of up to 20% have also been measured in DAFF trials. [29]

8.8 Nematodes and crown rot

The NGA has been involved in 22 field trials since 2007, in collaboration with NSW DPI, evaluating the impact of crown rot on a range of winter-cereal crop types and varieties. This work has greatly improved the understanding of crown rot impact and variety tolerance, but also indicates that we may be suffering significant yield losses from another 'disease' that often goes unnoticed.

Although the trials were not designed to focus on nematodes, a convincing trend was apparent after 2008 that indicated *P. thornei* was having a frequent and large impact on wheat variety yield.

These trials were designed to evaluate the effect of crown rot on variety yield and quality. However, they strongly suggest that *P. thornei* is also having a significant impact on yield performance. The results do not compare the levels of yield loss due to the two diseases but do indicate that there is a greater range in variety of *P. thornei* tolerance than currently exists for crown rot tolerance. [30]

8.8.1 Importance of variety choice

Variety choice appears a more valuable tool for use under *P. thornei* pressure than for crown rot management. It may be co-incidence, but four of the most widely adopted and successful wheat varieties in the northern grains region (EGA Wylie⫽, EGA Gregory⫽, Baxter⫽ and Sunvale⫽) are the varieties with the highest currently available level of *P. thornei* tolerance.

Root lesion nematodes are a 'disease' that has no obvious visual symptoms in the paddock. To improve management of this disease, growers must take more advantage of nematode testing. An increase in level of awareness of *P. thornei* status in individual paddocks and across properties will assist to:

- Develop sound hygiene practices to help limit further spread and reduce the risk of new infestations
- Provide a measure of the impact of varying management approaches designed to limit or reduce nematode build-up

This knowledge is also likely to provide direct economic gains from sound varietal and crop rotation choices. Soil testing for nematodes may also provide benefits in the identification of other plant parasitic species. [31]

29 K Owen, J Sheedy, N Seymour (2013) Root lesion nematode in Queensland. Soil Quality Pty Ltd Fact Sheet.

30 R Daniel (2013) Managing root-lesion nematodes: how important are crop and variety choice? Northern Grower Alliance/GRDC Update Paper, 16/07/2013,

31 R Daniel, S Simpfendorfer, G McMullen, John Thompson (2010) Root lesion nematode and crown rot – double trouble! Australian Grain, September 2010. http://www.ausgrain.com.au/Back%20Issues/203sogrn10/203sogrn10.pdf

SECTION 9

Diseases

A single disease management strategy rarely provides complete disease control. Using a number of integrated disease management techniques (IDM) is more likely to control diseases.

Controlling the major fungal diseases of chickpeas in the northern region requires an integrated approach to disease management and prevention. [1]

9.1 The diseases

Ascochyta blight. The pathogen survives and spreads in infected seed, stubble and on volunteers; chickpeas are the only known host in Australia. Fruiting bodies (pycnidia) of *Phoma rabiei* (also known as *Ascochyta rabiei*) develop on infected plant tissue, and spores, which ooze from wet pycnidia, are spread short distances and cause new infections. Under ideal conditions, Ascochyta blight can reproduce as fast as 5–7 days.

Botrytis grey mould (BGM). Similar to the Ascochyta blight pathogen, the BGM pathogen (*Botrytis cinerea*) can survive and spread in infected seed and stubble, and some strains produce dark, hard sclerotia, which also aid survival and spread. However, the BGM pathogen has a very wide host range, and is able to colonise dead and dying tissue of virtually any plant. Huge numbers of spores are produced on BGM lesions and are spread on air currents. BGM can also cycle in 5–7 days.

Phytophthora root rot (PRR). *Phytophthora medicaginis* survives as thick-walled oospores, which develop in infected roots of chickpea and other plants including lucerne and annual medics. When the soil is saturated with moisture, the oospores germinate to produce zoospores, which swim to and infect chickpea roots. The pathogen is spread by movement of infected soil and water.

Sclerotinia rot. Both *Sclerotinia* species (*S. sclerotiorum* and *S. minor*) survive as hard black sclerotia, in soil or mixed with seed. Both species have a very wide host range, including many weeds and most broadleaf crops. Infection of chickpea plants occurs directly at the crowns (both species) or from airborne spores produced in fruiting bodies on germinated sclerotia (*S. sclerotiorum*) (Table 1). [2]

1 GRDC (2013) Chickpea disease management. GRDC Resources Fact Sheet. May 2013, http://www.grdc.com.au/Resources/Factsheets/2013/05/Chickpea-disease-management

2 K Moore, M Ryley, T Knights, P Nash, G. Chiplin, G Cumming (2011) Chickpeas—varietal selection, paddock planning and disease management in 2011. Northern region (Goondiwindi). GRDC Update Papers April 2011

More information

http://www.grdc.com.au/Resources/Factsheets/2013/05/Chickpea-disease-management

M Fuhlbohm, M English, M Ryley, N Gannon, M Cahill (2004) Chickpea disorders: The Ute Guide. Department of Primary Industries and Fisheries, Queensland, and Grains Research and Development Corporation. http://www.grdc.com.au/GRDC-UteGuide-ChickpeaDisorders

http://grdc.com.au/Research-and-Development/DAN00143

K Moore, L Jenkins, P Nash, G Chiplin, S Bithell (2016), Chickpeas: What we learnt in 2015 and recommendations for 2016

P Matthews, D McCaffery, L Jenkins (2016), Winter crop variety sowing guide 2016 p85

K Moore, K Hobson, A Rehman (2014), Chickpea varietal purity and implications for disease management p138

K Moore, K Hobson, S Bithell (2016), Is chickpea on chickpea worth it?

Table 1: 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

Some of the disease terms that are useful to know when diagnosing chickpea diseases are pictured in Figure 1.

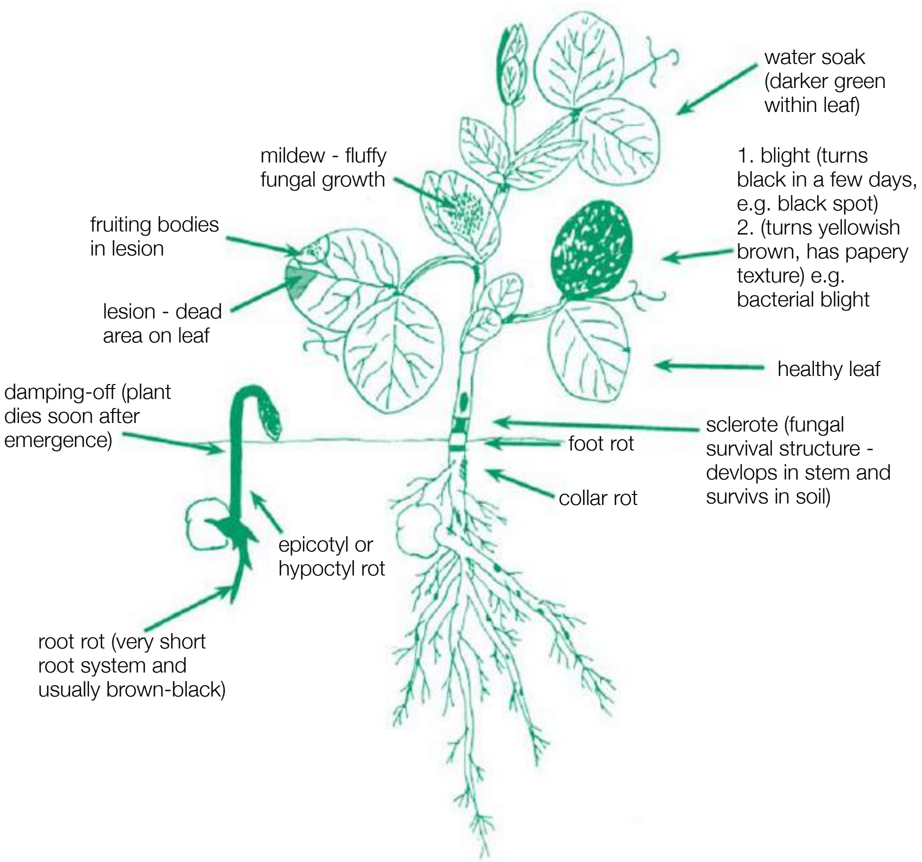

Figure 1: Chickpea disease diagnosis terms. (Source: Grain Legume Handbook).

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 (Figure 2). Diseases such as Ascochyta blight and PRR rot can cause total crop failures very quickly, whereas the effects of BGM and root-lesion nematodes on crop performance and yield may unfold more slowly.

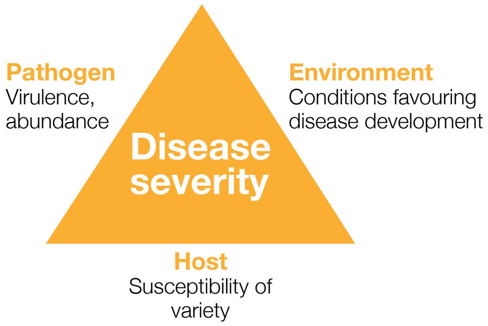

Figure 2: The virus and some bacterial disease triangle (Jones 2012).

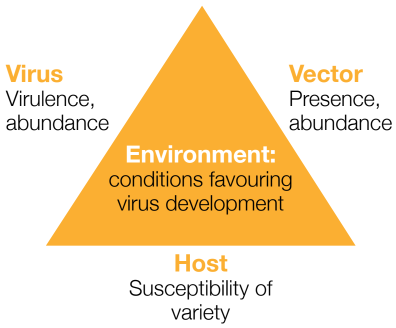

Figure 3: The disease triangle (Agrios 1988).

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 4 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.

More information

http://www.nga.org.au/results-and-publications/download/137/grdc-update-papers-diseases/botrytis-seedling-blight-in-chickpeas/grdc-adviser-update-paper-goondiwindi-march-2012.pdf

APVMA (2015), Permit to allow minor use of an agvet chemical product for the control of various fungal diseases in chickpeas and lentils

- **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.
- **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. Usually the virus spread has occurred by the time 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. [3]

More information

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/02/Developing-a-plan-for-chickpeas-2013

http://www.grdc.com.au/Media-Centre/Media-News/North/2013/06/Top-five-chickpea-challenges

Pulse Australia (2015), Chickpea: integrated disease management

Clean seed and care the recipe for chickpea success (2013)

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.

Paddock selection based on PRR is the first priority, followed by cropping history.

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. [4]

Integrated disease management (IDM) is an integrated approach of crop management to reduce chemical inputs and resolve ecological problems. Although originally developed for insect pest management, IDM programs now encompass diseases, weeds, and other pests.

Integrated disease management 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:

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

4 Pulse Australia (2011) Chickpea Integrated Disease Managment. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/idm-strategies

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 (e.g. removal of diseased plants to prevent spread of infection). 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, for example, 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 life cycle.
- 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.

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* and *Phytophthora medicaginis*.

1. Rotation

- Develop a rotation of no more than 1 year of chickpea in 4 years.
- Plant chickpea into standing stubble of previous cereal or sorghum stubble 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*, *Rhizoctoni*a and *Phytophthora medicaginis*.
- *Ascochyta rabiei* is chickpea-specific, whereas *Botrytis cinerea* has a wide host range including sunflower, bean, pea, and weeds (e.g. *Euphorbia* spp., groundsel and emu-foot).
- Lucerne, medics and chickpea are hosts for *Phytophthora medicaginis*, and *Phoma medicaginis* var. *pinodella* can be hosted by lucerne, clover, field pea, lupin and chickpea as well as *Phaseolus* spp.

2. History of chickpea diseases

- Previous occurrence of soil-borne diseases (PRR, Sclerotinia stem rot or *Pratylenchus* nematodes) constitutes a risk for subsequent chickpea crops for up to 10 years.

More information

http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0003/431274/Moore-Chickpea-2011-Crops-and-Recommendations.pdf

- At least 500 m from the previous year's chickpea crop.

3. Weeds

- Realise that 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 or sulfonylurea 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 2 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 about 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

9.4.4 Foliar fungicides

Foliar fungicides are essential for the management of Ascochyta blight in all varieties, and are an important tool for the management of BGM. Varieties with higher levels of

Ascochyta blight resistance do not require as many sprays as susceptible varieties. The success of foliar fungicides depends on timeliness of spraying (hence the importance of regular crop monitoring), appropriate fungicide selection, and correct application (Table 2). Early detection and fungicide application is vital.

Table 2: Foliar fungicides for the control of Ascochyta blight and Botrytis grey mould in chickpea

✓ Registered product label claim

Active ingredient:	Carbendazim	Chlorothalonil	Mancozeb
Example trade name:	Spin Flo®	Barrack®/Unite® A	Dithane® Rainshield
Ascochyta blight		✓	✓
Botrytis grey mould	✓		✓
Damping-off (Kabulis)			
Phoma root rot			
Phytophthora root rot			
Jurisdiction	All states	All states	All states

[A]These are the only registered chlorothalonil products. It is an offence to use any other product.

Refer to the current product label for complete ‘Directions For Use’ prior to application.

Prior to the use of any crop protection product, ensure that it is currently registered or that a current permit exists for its use in chickpeas.

More information

Registered labels and current permits can be found on the APVMA website: www.apvma.gov.au.

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Chickpea-varieties-selecting-horses-for-courses

9.5 Ascochyta blight

9.5.1 Background

Ascochyta blight, caused by the fungus *Ascochyta rabiei* (also known as *Phoma rabiei*), is a serious disease of chickpeas in Australia. The fungus can infect all aboveground parts of the plant and is most prevalent in areas where cool, cloudy and humid weather occurs during the crop season.

Ascochyta blight first caused widespread damage to chickpeas in Australia in 1998 when extremely wet conditions favoured disease development and spread. Ascochyta blight is now considered endemic in all growing regions of Australia. Unlike some insect-control strategies, there is no economic threshold for Ascochyta blight. Management strategies are aimed at preventing the occurrence of disease and limiting its spread.

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.

9.5.2 Economic importance

This disease is very serious, as it has caused severe damage and losses in chickpeas. In the very wet winter of 2010, many crops in north-west New South Wales (NSW) were wiped out completely by Ascochyta blight.

Biology and epidemiology

Ascochyta blight causes economic losses only on chickpea. There are no other known hosts of the pathogen in Australia, but different *Ascochyta* species infect faba beans, lentils and field peas. The pathogen survives between seasons on infected plant residues, on infected or contaminated seed and on infected volunteer chickpea plants (Figure 4).

More information

[K Moore, K Hobson, P Sambasivam, R Ford, S Harden, P Nash, G Chiplin, S Bithell (2015), Chickpea ascochyta - is the pathogen changing and what are the implications for management]

[K Moore, K Charleston (2014), Ascochyta infected chickpea area widens in the north]

[K Moore, K Hobson, N Dron, S Harden, S Bithell, P Sambasivam, R Ford, Y Mehmood, J Davidson, S Sudheesh, S Kaur (2016), Chickpea Ascochyta: latest research on variability and implications for management]

[K Moore, K Hobson, S Harden, P Nash, G Chiplin, S Bithell (2015), Effect of chickpea ascochyta on yield of current varieties and advanced breeding lines.]

[K Moore, M Ryley, G Cumming, L Jenkins (2015), Chickpea: Ascochyta blight management]

[K Moore, K Hobson, S Harden, P Nash, G Chiplin, S Bithell (2015), Effect of chickpea Ascochyta on yield of current varieties and advanced breeding lines – Tamworth 2014 p89]

[K Moore, K Hobson, S Harden, P Nash, G Chiplin, S Bithell (2015), Chickpea ascochyta - evidence that varieties do differ in susceptibility of pods]

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.

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 3 years in the paddock.

Ascochyta blight can develop over a wide range of temperatures (5–30°C) and needs only 3 h of leaf wetness to infect. 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.

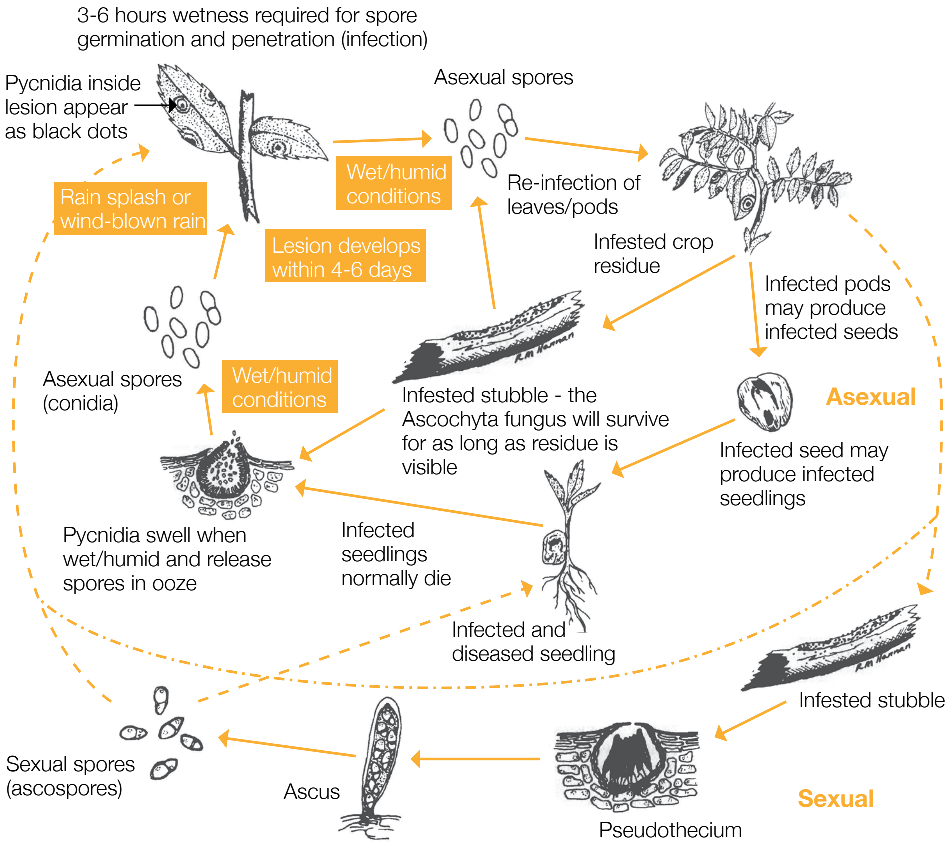

Figure 4: Life cycle of Ascochyta blight pathogen. Note: Only the asexual phase is known to occur in Australia at this time.

9.5.3 Symptoms

Ascochyta blight infects the leaves, stems and pods of chickpea plants, causing tan/brown, rounded lesions on affected plant parts.

Symptoms become visible in 7–10 days as a pale green–yellow discoloration on leaves, often referred to as 'ghosting' (Figure 5).

Toward the centre of the lesion, small, black fruiting bodies called pycnidia develop in 10–14 days, often in concentric rings (Figure 6). Spores ooze out of pycnidia and are spread by rain-splash upwards within the plant and sideways to nearby healthy plants.

Lesions often girdle the stems of the plant, causing them to weaken and subsequently break off, making later detection difficult (Figure 7). Circular 'hot spots' or 'foci' consisting of plants with severe infection can appear in crops, but by this stage considerable damage has occurred. Seeds can become infected after lesions develop on pods (Figure 8).

Figure 5: Ascochyta blight: leaf ghosting may appear 7–10 days after infection following rainfall or heavy dew.

Figure 6: Note the concentric circles of brown–black dots in the centre of the lesions. These are the pycnidia or fruiting bodies, which are unique to Ascochyta blight.

Figure 7: 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.

Figure 8: Pod lesions are similar in appearance to leaf lesion. They lead to infection of the seed. Do not keep planting seed from any crop that has been identified as having Ascochyta blight. (Photos: G. Cumming, Pulse Australia)

9.5.4 Management options

Follow the principles of IDM, which include:

- crop rotation and paddock selection
- reducing proximity to previous season chickpea stubble
- growing resistant varieties
- using clean seed and fungicide seed dressings
- regular crop monitoring
- strict hygiene on and off farm
- strategic use of foliar fungicides

Note: Chickpea seed dressings protect only 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*.

Differing spray programs have been developed based on each variety's Ascochyta blight rating.

Chickpea Ascochyta blight 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 blight spray program is regular monitoring combined with timely application of registered fungicides.

Resistant (R): Genesis™ 090, Genesis™ 425

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.

Monitor the crop 10–14 days after each rain event.

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.

Moderately resistant (MR): PBA HatTrick⛣, PBA Boundary⛣, Genesis™ 114

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.

Monitor the crop 10–14 days after each rain event.

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 (S): Jimbour⛣, Kyabra⛣, PBA Pistol⛣

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.

Mancozeb is often the preferred fungicide for these first two applications because it can be applied with a Group A grass herbicide.

Continue monitoring 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 or longer since the last application, spray again just before the next rain event.

A fungicide program

A fungicide program needs to account for several factors.

Disease risk categories

Based on:

- varietal susceptibility or resistance
- source of seed and treatment of seed
- planting proximity to chickpea crops of previous seasons
- level of *Ascochyta* inoculum present from crop residue or volunteer plants
- climatic conditions in relation to disease infection

Registration status

The product must be registered or have a permit for the disease and use.

Withholding period

All products and timings used in the fungicide program must meet Australian withholding periods and export slaughter intervals to satisfy overseas markets.

Fungicide resistance management

The maximum number of sprays of a product must be adhered to, in order to minimise the risk of fungicide resistance developing.

Mode of action

Using products with a range of mode of actions for control of diseases further reduces the chance of fungicide resistance development and improves efficacy. Fungicides are also recommended at times of the disease life cycle where they will be most effective according to their mode of action.

Early harvest

Harvest at maturity to minimise *Ascochyta* seed infection and potential down grading. Seed damage from Ascochyta blight is usually more severe when crops are harvested late. Moisture content allowable on delivery is 14%. Harvest losses, seed splitting and downgrading in quality can be substantial if chickpea is harvested at below 12% moisture.

More information

[M Ryley, K Moore, G Cumming, L Jenkins (2015), Chickpea: Managing Botrytis grey mould]

9.6 Botrytis grey mould

9.6.1 Background

Botrytis grey mould in chickpea is caused by the fungus *Botrytis cinerea*, 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; however, *B. cinerea* does not infect cereals or grasses.

Botrytis cinerea has been recorded on over 138 genera of plants in 70 families. Legumes and asteraceous plants comprise about 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 blight, which is more dependent on inoculum, at least in the early phases of an epidemic.

Botrytis cinerea also causes pre- and post-emergent seedling death. This happens when chickpea seed, infected during a BGM outbreak, is used for sowing. This seedling disease does not need the wet conditions that favour BGM.

9.6.2 Economic importance

Botrytis grey mould 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.

9.6.3 Biology and epidemiology

Botrytis cinerea produces diffuse, white fungal growth, which later turns grey due to the production of huge numbers of spores borne in clusters at the ends of dark stalks.

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.

Botrytis grey mould is favoured by moderate temperatures (20–25°C) and frequent rainfall events. It does not become a risk until the average daily temperature is ≥15°C. 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. cinerea*.

The pathogen can survive on and in infected seeds, in infected stubble, on alternative hosts, in dead plant tissue and as sclerotia. 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 Western Australia suggests that sclerotia of *B. cinerea* may not be able to survive over summer because they lose their viability during hot weather.

Irrespective of its mode(s) of survival, the experience of the 2010 chickpea crop indicates that under conducive conditions, BGM can develop rapidly not only within crops, but also across districts and regions.

9.6.4 Symptoms

Often, the first symptom of BGM infection in a crop is drooping of the terminal branches. If groups of plants are infected, these may appear as yellow patches in the crop (Figure 9).

The diagnostic feature is a grey 'fuzz' (Figure 10), which under high humidity, develops on flowers (Figure 11), pods (Figure 12), stems and on dead leaves and petioles. Lesions can develop anywhere along the stem, but are usually first found on the lower part of the stems often starting in leaf axils.

Infected seeds are usually smaller than normal and are often covered with white to grey fungal growth (Figure 13).

Figure 9: If groups of chickpea plants are infected these may appear as yellow patches in a crop.

Figure 10: The diagnostic feature for Botrytis grey mould is a grey 'fuzz', which develops under high humidity.

Figure 11: Botrytis grey mould on chickpea flowers.

Figure 12: Botrytis grey mould on pods.

Figure 13: Botrytis grey mould on seed. (Photos: G. Cumming, 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. Small, dark brown–black resting bodies (sclerotia) of *B. cinerea* may develop on infected dead tissue, and are capable of producing spores on their surface.

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 sclerotia develop.

By contrast, the sclerotia of *B. cinerea* are more rounded and they usually develop after the stems die. They are smaller than the sclerotia of *S. sclerotiorum*, but larger than the angular sclerotia of *S. minor*.

9.6.5 Management options

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 in the air 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.

Volunteer control: the 'green bridge'

Volunteer chickpea plants growing in or near paddocks where BGM was a significant problem are a likely method of carry-over and must be managed by application of herbicide or cultivation.

This will also reduce carryover of the Ascochyta blight pathogen.

Seed source

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.

Seed fungicides (dressings)

Thiram-based fungicide seed dressings are effective in significantly reducing, but not eliminating, BGM from infected seed.

Paddock selection

Paddocks in which chickpeas were affected by BGM should not be re-sown to chickpea, faba bean or lentil the following season. Irrespective of disease, the paddock should not be re-sown to chickpeas for at least 3 years. Nor should chickpea be sown beside paddocks where BGM was an issue the previous season.

As is the case 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.

Sowing time and row spacing

If long-term weather forecasts suggest a wetter than normal year (La Niña), consider sowing in the later part of the suggested sowing window for your district and on wider rows (e.g. 100 cm); the latter results in increased air movement through the crop and reduced humidity within the canopy.

More information

http://www.publish.csiro.au/paper/AR06120.htm

http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0004/431266/Chickpea-seed-tests.pdf

Varietal resistance

All current commercial varieties suitable for the southern and western regions are susceptible to BGM.

Foliar fungicides

In seasons and situations favourable to the disease, a preventative spray of a registered fungicide immediately prior to canopy closure, followed by another application 2 weeks later, will assist in minimising BGM development in most years. This is particularly important, as often the seasons favourable to BGM will result in large crop canopies. This makes penetration of foliar fungicides very poor once the canopy has closed over.

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.

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, initial timely and thorough application is critical.

9.7 Phytophthora root rot

Phytophthora root rot is not a major chickpea disease in southern and Western Australia, but it is a major disease north of central NSW and Queensland.

9.7.1 Background

Phytophthora root rot is a disease of chickpea caused by the fungus-like oomycete *Phytophthora medicaginis*, which is widespread in the cracking clay soils of northern NSW and southern Queensland. It can cause significant yield losses (Figure 14) in wetter than normal seasons or following periods of soil saturation in normal seasons. Lucerne, perennial and annual medics (*Medicago* spp.), and other leguminous plants including sulla (*Hedysarum* spp.) and sesbania (*Sesbania* spp.) can also host *P. medicaginis*.

Figure 14: Cultivated areas were killed by Phytophthora. Only plants on top of contours survived. (Photo: M. Schwinghamer, NSW DPI)

More information

K Moore, K Hobson, S Harden, G Chiplin, S Bithell, L Kelly, W Martin, K King (2016), Phytophthora in chickpea varieties HER15 trial –resistance and yield loss

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

K Moore, M Ryley, M Schwinghamer, G Cumming, L Jenkins (2015), Chickpea: Managing Phytophthora root rot

K Moore, T Knights, S Harden, P Nash, G Chiplin, K Hobson, M Ryley, W Martin, K King (2014), Response of chickpea genotype to Phytophthora root rot (Phytophthora medicaginis) – Warwick Qld 2013 p136

K Moore, L Kelly, M Ryley, K Hobson, T Knights, S Harden, W Martin, K King, P Nash, G Chiplin, S Bithell (2015) Phytophthora in chickpea varieties - resistance rankings and yield loss.

K Moore, M Ryley, K Hobson, T Knights, S Harden, W Martin, K King, P Nash, G Chiplin (2014), Phytophthora tolerance in chickpea varieties.

Figure 15: Phytophthora in a water-course. (Photo: M. Schwinghamer, NSW DPI)

Phytophthora and waterlogging (where roots die from low oxygen levels) are 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 (Figure 15), or where the soil has been compacted or has hard pans.

However, under very wet conditions, entire paddocks can be affected.

9.7.2 Economic importance

Phytophthora root rot is a serious disease of chickpeas in southern Queensland and northern NSW.

Although no economic losses have been reported in the southern regions, it remains a potential threat in areas with lucerne, medics or heavy textured soils.

9.7.3 Biology and epidemiology

Phytophthora medicaginis survives in soil mainly as thick-walled oospores (Figure 16), 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.

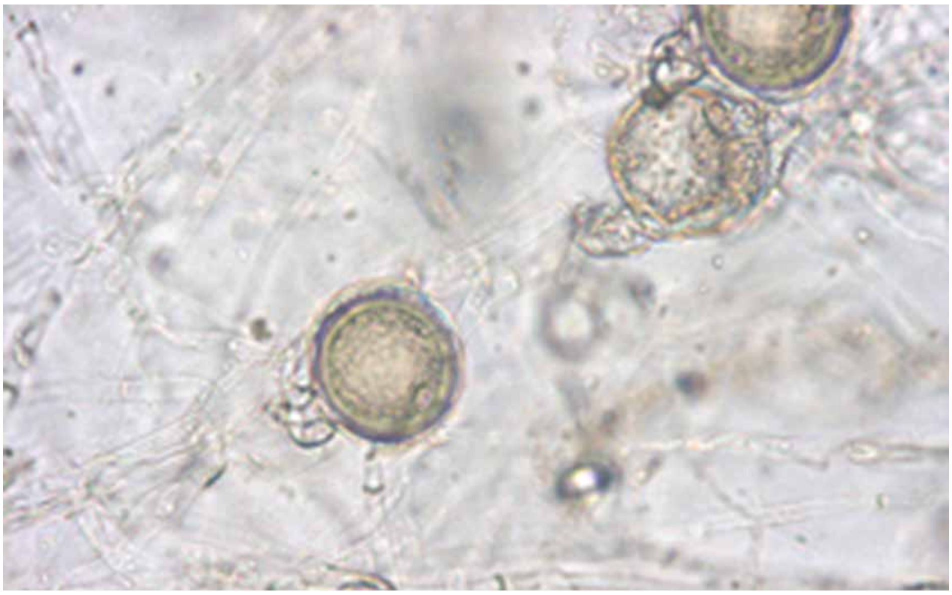

Figure 16: Phytophthora medicaginis oospores can survive in soil for up to 10 years. (Photo: G. Chiplin, NSW DPI)

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.

9.7.4 Symptoms

Infection by *P. medicaginis* can occur at any growth stage, causing seed decay, pre- and post-emergence damping off, loss of lower leaves (Figure 17), and yellowing, wilting and death of older plants.

Symptoms are sometimes delayed if temperatures are cool and the soil is moist. On young plants the lesions may extend up the stem for 10 mm or more above ground level (Figure 18). Lateral roots and tap root die (Figure 19) or dark brown/black lesions often girdle the taproots.

Figure 17: Severely affected plants (left) have no lateral roots and defoliation below tips of stems. (Photo: J. Wessels, Qld Gov.)

Figure 18: Basal lesions extending up the plant stem. (Photo: M. Ryley, Qld Gov.)

Figure 19: Phytophthora root rot-affected plant (right) with lateral and tap root death. (Photo: M. Ryley, Qld Gov.)

Figure 20: New roots forming from the top of the taproot (Phytophthora root rot). (Photo: M. Fuhlbohm, Qld Gov.)

Plants with PRR 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 (Figure 20).

Symptoms of waterlogging can be confused with those of PRR (Table 3), but differ in the following ways:

- Plants are most susceptible to waterlogging at flowering and early pod-fill.
- Symptoms develop within 2 days of flooding, compared to at least 7 days for PRR.
- 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.

Table 3: Differences between Phytophthora root rot 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 after a week or more	Symptoms onset quite rapid
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 and varietal choice	Manage through paddock selection, no irrigation in reproductive phase

9.7.5 Management options for PRR

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 blight and BGM. Thus, PRR can only be managed by pre-sowing decisions and assessing risks for individual paddocks.

Development of the disease requires 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 not to sow chickpeas in high-risk paddocks, which are those with a history of:

- PRR noted in previous chickpea or lucerne crops
- lucerne or annual or perennial medics
- waterlogging or being flood-prone

However, if you choose to sow chickpeas in high-risk paddocks, the following measures will reduce losses from PRR:

- Grow 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. Current commercial varieties differ in their resistance to P. medicaginis, with Yorker and PBA HatTrick having the best resistance and are rated MR (historically Yorker has been slightly better than PBA HatTrick), while Jimbour is MS-MR, Flipper and Kyabra are MS and PBA Boundary has the lowest resistance (S). PBA Boundary should not be grown in paddocks with a history of PRR, lucerne, medics or other known hosts such as sulla.

More information

http://www.grdc.com.au/Media-Centre/Ground-Cover/Ground-Cover-Issue-104-May-June-2013/Overcoming-Phytophthora-root-rot-in-chickpeas

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/02/Developing-a-plan-for-chickpeas-2013

- Although registered for use on chickpeas, metalaxyl seed treatment is expensive, does not provide season-long protection and is not recommended. [5]

9.7.6 Management options for waterlogging

- Avoid poorly drained paddocks and those prone to waterlogging (Figure 21).
- 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.

Figure 21: Waterlogged crop areas seen from the air.

9.8 Sclerotinia stem and crown rot

9.8.1 Background

Sclerotinia stem and crown rot of chickpea are caused by *Sclerotinia* spp. Three species of *Sclerotinia* are reported to cause the disease *S. sclerotiorum*, *S. minor* and *S. trifoliorum*. Of these, the most common is *S. Sclerotiorum.*

In 2010, Sclerotinia was more common than in previous years and in some paddocks caused serious damage, including 100% loss in one Kabuli crop near Dubbo (Figure 22).

[5] K Moore, K Hobson, S Harden, G Chiplin, S Bithell, L Kelly, W Martin, K King (2016), Phytophthora in chickpea varieties HER15 trial –resistance and yield loss. https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/03/Phytophthora-in-chickpea-varieties-HER15-trial-Resistance-and-yield-loss

Figure 22: Sclerotinia sclerotiorum killed these plants; eventually the whole crop was lost. (Photo: K. Moore, NSW DPI)

More information

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/02/Sclerotinia-stem-rot-Managing-the-disease-in-2013

9.8.2 Economic importance

Sclerotinia can cause severe damage in chickpeas. This has occurred in Kabuli chickpeas in Victoria.

9.8.3 Biology and epidemiology

Among the three *Sclerotinia* spp., *S. sclerotiorum* has the widest reported host range. It infects >400 plant species, whereas *S. minor* causes diseases on crops in at least 53 plant genera and significant economic losses in peanut, sunflower and lettuce. *Sclerotinia trifoliorum* causes diseases on plants belonging to 21 plant genera and major economic losses in legumes, particularly forage legumes such as *Medicago* spp. and *Trifolium* spp.

All three species can survive in soil as sclerotia for 10–12 years without susceptible host plants. The crown can be infected by any of the three species, although usually one species dominates in a particular field.

The disease is favoured by cool, moist weather. Once established, the fungus can move rapidly to neighbouring healthy, tissue. A few days after infection, plants start to wither and die. The fungus is carried over to the next year in the infected plants. It is suspected, on the basis of optimal growth temperatures, that *S. trifoliorum* prefers cooler conditions than *S. sclerotiorum*.

9.8.4 Symptoms

Sclerotinia appears mainly on older plants. At first water-soaked patches (lesions) appear on the stems and leaves, and later affected areas develop a soft, slimy rot which exude droplets of brown liquid. The infected tissues dry out and they become covered with a fine white web of fungus growth. Small black spots, irregular in size and shape may sometimes be seen just below the surface, mingled with the white fungus growth.

Later on, stem lesions turn grey, the white growth disappears and the branch above the lesion dies. Affected plants wilt and die rapidly, without losing their leaves.

A late infection can affect the pod and seeds. Infected seeds are smaller than normal and discoloured.

Sclerotinia sclerotiorum is the prevalent species in cooler, wetter regions, whereas *S. minor* is more common in warmer drier environments. Both species cause a basal stem rot when their sclerotia germinate in soil and infect the base of the plant.

Sclerotinia sclerotiorum (Figure 23) and *S. trifoliorum* produce large, irregular-shaped sclerotia 5–10 mm in diameter, as high up as 20–30 cm on the stem.

Figure 23: Sclerotia of S. sclerotiorum. (Photo: M. Ryley, Qld Gov.)

Figure 24: Sclerotia of S. minor. (Photo: K. Moore, NSW DPI)

Sclerotinia minor (Figure 24) produces sclerotia that are angular and much smaller, rarely larger than 2–3 mm in diameter.

In dense crops, during moist conditions, a white cottony fungal weft develops around the base of plants (Figure 25).

Figure 25: Fungal weft of Sclerotinia in the lower canopy. (Photo: K. Moore, NSW DPI)

Under cool wet conditions, *S. sclerotiorum* sclerotia can germinate to produce small cup like structures (apothecia) at ground level (Figure 26).

These release air-borne ascospores that infect aboveground parts of the chickpea plant, often starting in leaf axils (Figure 27).

Stem tissues above and below the infection point initially remain green.

Figure 26: Apothecia are produced at ground level.

Figure 27: Ascospore infection of chickpea stem by S. sclerotiorum. (Photo: G. Cumming, Pulse Australia)

9.8.5 Management options

Both *S. sclerotiorum* and *S. minor* have wide host ranges including many broadleaf weeds and crops such as canola, faba bean and sunflower.

Cotton and cereals are not hosts to either species.

Reduce the risk of losses from Sclerotinia rot by sowing seed free of sclerotia and by not sowing chickpea in paddocks that have had alternative host crops in the past 10 years, because the resting structures (sclerotia) can survive for that long.

It is acknowledged that 10 years, in most situations, is impractical, but do not sow chickpea in paddocks that had a broadleaf crop (other than cotton) last season.

The disease risk can be reduced by using disease-free seed. It is also important to avoid sowing chickpeas on areas where the disease is known to be present. If severe infection occurs, the area should be burnt and ploughed deeply to kill the fungus in the soil.

Crop rotation will reduce the risk of infection. Cereals, which are not a host, should be grown for several seasons before returning to chickpeas or other pulses or canola. Other hosts to *Sclerotinia* are the oilseed crops (e.g. canola, pulses and broadleaf weeds such as capeweed).

9.9 Root rots including damping off (*Fusarium*, *Rhizoctonia* and *Pythium* spp.)

9.9.1 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 (Figure 28).

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.

9.9.2 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.

9.9.3 Disease cycle

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.

9.9.4 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.

Figure 28: Rhizoctonia root rot. Optimum soil temperature is 24–26°C; disease is worse on light sandy soils.

9.10 Collar rot (*Sclerotium rolfsii*)

9.10.1 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, particularly in central Queensland, 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 (Figure 29); 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 (about 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.

9.10.2 Economic importance

Collar rot is generally a minor disease in chickpea. However, the disease has been particularly severe in irrigated Macarena (Kabuli).

9.10.3 Disease cycle

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.

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.

Figure 29: Webs of Sclerotium rolfsii mycelium at the base of an infected chickpea plant.

9.11 Virus management

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 aphids that fly in to crops do not stay long and do not normally colonise plants. Typical virus symptoms are bunching, reddening, yellowing, death of shoot tips and early death of whole plants. However, it should be remembered that none of these are diagnostic for viruses.

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 uniform closed canopy and controlling weeds (Schwinghamer *et al.* 2009). Seed and foliar insecticides are not recommended for chickpea viruses. [6]

Virus management in pulses aims at prevention through integrated management practice that involves controlling the virus source, aphid populations and virus transmission into and within pulse crops.

Rotate pulse 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 that are a green bridge host for viruses and a refuge for aphids and their multiplication.

Aphids are the major means by which viruses enter chickpea crops. Winged aphids acquire viruses by feeding on alternative hosts (particularly lucerne) before landing on chickpeas. They feed briefly, thus transmitting viruses, and then fly on. Cucumber mosaic virus (CMV) and Alfalfa mosaic virus (AMV) are non-persistently transmitted by a range of aphid species. *Aphis gossypii* is one of many possible vectors of both. The luteoviruses are persistently aphid-transmitted, but are more vector-specific.

6 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

Aphids seldom colonise chickpeas or move between adjacent plants to feed. The result is that chickpea crops show a characteristic scattered distribution of individual virus-infected plant (Figure 30). This contrasts with crops such as peas and faba beans, which aphids do colonise, and patches of infected plants are common in these crops. The pea aphid (*Acyrthosiphon pisum*) appears to colonise chickpeas more readily than other aphid species and is likely to be an important vector.

Figure 30: Scattered appearance of virus-infected plants. (Photo: M. Schwinghamer, NSW DPI)

Aphid activity is influenced by seasonal conditions and will require early monitoring in nearby crops and pastures.

Control measures for viruses in chickpea are not adequate at present. Application of seed and foliar insecticides, aimed at preventing feeding by aphids, has failed to prevent infection by viruses in field experiments. [7]

Best agronomic management can help to reduce damage by viruses and includes:

- Retain 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.
- Plant on time and at the optimal seeding rate. These practices result in early canopy closure, which reduces aphid attraction (see Figure 31).
- Ensure adequate plant nutrition.
- Control in-crop, fence line and fallow weeds. This removes in-crop and nearby sources of vectors and virus.
- Avoid planting adjacent to lucerne stands. Lucerne is a perennial host on which legume aphids and viruses, especially AMV and Bean leaf roll virus (BLRV), survive and increase.
- Seed treatment with insecticides (e.g. imidacloprid) are 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 Beet western yellows virus (BWYV) sometimes found in canola, consider growing chickpeas (and other pulse crops) away from canola. [8]

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

8 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

More information

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/02/Developing-a-plan-for-chickpeas-2013

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/02/Virus-in-chickpea-in-northern-NSW-2012

http://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2010/05/Chickpeas-in-2010-Viruses-in-2009-Recommendations-for-2010

M Sharman, K Moore, J van Leur, M Aftab, A Verrell (2014), Impact and management of viral diseases in chickpeas

K Moore, A Verrell, M Aftab (2014), Reducing risk of virus disease in chickpeas through management of plant density, row spacing and stubble

Pulse Australia (2015), Managing viruses in pulses

A Verrell, K Moore (2015), Managing viral diseases in chickpeas through agronomic practices

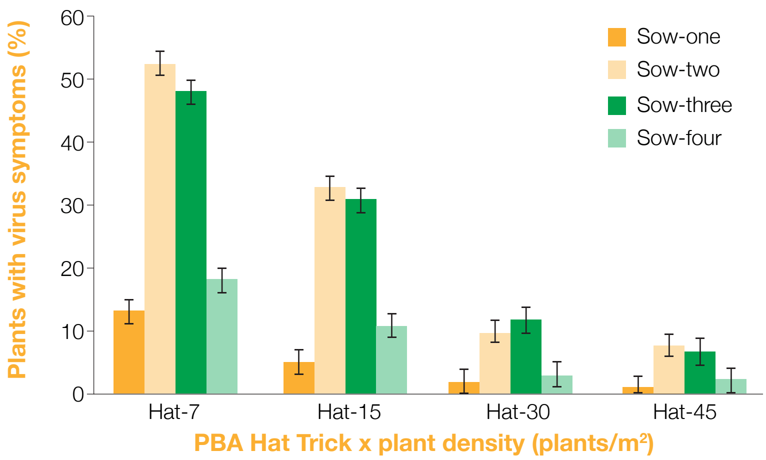

Figure 31: Proportion of plants with virus symptoms for sowing date by plant density for PBA HatTrick⊕. [9]

9.11.1 The northern experience in 2013

In 2013, virus infection was found in almost all chickpea crops inspected from southern Queensland to Wellington, NSW. The incidence of virus infection was generally lower than observed in 2012, with most crops inspected having <5% plants with symptoms, but it was as high as 30–50% in several crops from the Breeza–Werris Creek area and Edgeroi, NSW. Overall, the most prevalent virus was BWYV, and in some locations, >90% of symptomatic plants were infected with BWYV (Table 4).

Related virus species also react with the BWYV assay, so it is likely there was a mix of BWYV-like viruses present at many locations. Some of the main outcomes from the chickpea surveys in northern NSW were:

- A higher proportion of BWYV infections was found at, and north of, the Liverpool Plains. Higher proportion of AMV infections in the south (Table 4). Very low levels of BLRV and CMV.
- Up to 15% of non-symptomatic plants from the Liverpool plains still had BWYV infection.
- Accurate identification by polymerase chain reaction (PCR) has shown the aphid-transmitted *Luteovirus* species to have a wide geographical range in a number of alternative weed hosts (Table 5).
- Soybean dwarf virus (SbDV) was the major virus affecting several crops in the Edgeroi region in October 2013 and was confused with BWYV in the antibody test (Table 4).

[9] 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

Table 4: Percentage infection of Beet western yellows virus (BWYV), Alfalfa mosaic virus (AMV), Bean leaf roll virus (BLRV) and Cucumber mosaic virus (CMV) from chickpeas displaying virus symptoms in northern NSW as determined by tissue blot immunoassay (TBIA) diagnostic

Virus identification was based on antibody reaction. Sample locations shown roughly from north to south. Note that the BWYV infections may be a complex of related viruses. Samples from most locations were also tested for Turnip mosaic virus (TuMV), but no positives were detected. n.t., Not tested

Location	No .of plants tested	% BWYV	% AMV	% BLRV	% CMV
Boomi	6	100	0	n.t.	n.t.
North Star	12	67	17	n.t.	n.t.
Moree	19	79	0	0	0
Edgeroi	32	62	0	n.t.	n.t.
Edgeroi	17	47	0	n.t.	n.t.
Tamworth	15	60	20	n.t.	0
Tamworth	30	87	10	n.t.	0
Breeza	18	89	0	5	6
Breeza	25	88	8	4	8
Breeza	26	77	0	0	0
Breeza	19	53	5	0	5
Liverpool Plains	20	90	10	5	0
Liverpool Plains	21	90	10	5	0
Werris Creek	15	73	13	n.t.	0
Pine Ridge	15	93	7	n.t.	0
Pine Ridge	15	80	13	n.t.	0
Blackville	15	13	67	n.t.	0
Gilgandra	14	7	78	n.t.	n/t
Gilgandra	38	21	71	0	3
Gilgandra	49	12	88	0	2
Wellington	30	10	73	n/t	n.t.
Wellington	16	19	63	0	0
Wellington	15	7	60	0	0
Wellington	20	5	55	0	0

Biology of significant viruses of pulses, particularly chickpeas

Accurate identification of viruses is critical for the long-term success of resistance breeding and for meaningful studies of how viruses survive in weed hosts and move into crops. To this end, improved accurate diagnostics are being developed for the luteoviruses, to help overcome uncertainty of virus identifications that can result from cross-reactions of viruses to some antibodies. PCR has been used for BWYV, BLRV, Phasey bean virus (PhBV) and SbDV to investigate the host range of the virus species from various locations (Table 5). Although testing continues, marshmallow weed is commonly found infected with BWYV from many locations and burr medic is a host for BLRV, PhBV and SbDV.

Table 5: Identification of virus species in different plant hosts from different locations in the northern region confirmed by species-specific polymerase chain reaction (PCR)

Testing of selected samples from 2012 and 2013 surveys

Virus (by PCR or sequencing)	Plant host	Locations
Beet western yellows virus	Chickpea	Wellington, Breeza, North Star, Boomi
	Canola	Ardlethan, Burren Junction, Bellata
	Marshmallow	Wagga Wagga, Coolamon, Griffith, Hillston, Leeton, Narrandera, Wellington, Tamworth, Narrabri, Wee Waa, North Star, Goondoowindi, Grantham
	Turnip weed	Gravesend, Wee Waa, Burren Junction
	Sonchus sp. Coolamon Shepherds purse	Kingsthorpe, Boomi
Bean leaf roll virus	*Chickpea*	*Wellington, Edgeroi*
	Burr medic	Wellington
Phasey bean virus	Chickpea	Kingsthorpe, Boomi, North Star, Edgeroi, Burren Junction, Breeza, Horsham
	Faba bean	Edgeroi
	Burr medic	Boomi, Burren Junction, Wee Waa
	Lentil	Breeza
	Vetch	Kingsthorpe
Soybean dwarf virus	Chickpea	Wellington, Gilgandra, Breeza, Edgeroi, Bellata, North Star, Boomi, Clifton

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 (Verrell 2013, Moore *et al.* 2014). 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 (Verrell 2013).

Inter-row planting into standing wheat stubble significantly reduced virus incidence in small trial plots of PBA HatTrick⍉ compared with the same amount of stubble slashed low to the ground (Moore *et al.* 2014). 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 (Hawthorne 2008; Verrell 2013, 2014), 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⍉ and PBA HatTrick⍉, although both these varieties have been observed with high rates of infection under high disease pressure. 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 (van Leur *et al.* 2014), the sometimes high incidence of BWYV in canola indicates it may be prudent to avoid planting chickpea and other pulse crops next to canola. [10]

Proximity to canola plantings

Larger canola plantings may have been responsible for unusually severe outbreaks of viruses in the northern region's chickpea crops in 2012.

Plant pathologists say the link is unproven, but canola crops host BWYV, which could spread to chickpeas.

Canola and turnip weed close to surveyed chickpea paddocks showed high infections of BWYV and Turnip mosaic virus (TuMV) and could have played a role in the virus epidemic in chickpeas. The outbreak of BWYV was especially severe on the Liverpool Plains, NSW, and was costly as it wiped out several chickpea crops.

In 2012, the area sown to canola was about five times the long-term average because of better prices for canola and poor prices for other crops. Canola is sown earlier in the season than chickpeas. BWYV inoculum could build up in canola over winter then spread to chickpeas in spring and cause severe yield losses.

Growers need to pay attention to the whole farming system and growing environment of their crops to ensure that plants are healthy enough to fight incursions. Viruses are more severe in poor growing paddocks.

A healthy plant seems to have the ability to withstand the virus, so it is worth following recommended agronomic practices to reduce the chance of virus infection and increase the ability of plants to resist the virus. This includes sowing in standing stubble because virus-spreading aphids tend to be more attracted to plants that are in poor growing paddocks or growing in bare ground.

Researchers try to identify resistance as part of the breeding program in order to deliver varieties with improved resistance. [11]

9.11.2 Economic importance

Plant viruses occur in all states; however, they are a significant problem in chickpeas in northern NSW and southern Queensland, where total crop failures have occasionally occurred.

The damage caused by the viruses varies greatly from season to season and depends on the prevalence of aphids.

9.11.3 Virus types

Viruses that cause significant losses in chickpea include the following.

Luteoviruses:

- Bean leaf roll virus (BLRV)
- Beet western yellows virus (BWYV)
- Subterranean clover red leaf virus (SCRLV)
- Subterranean clover stunt virus (SCSV)

Shoot tip virus complex:

- Alfalfa mosaic virus (AMV)
- Cucumber mosaic virus (CMV) (Figures 31 and 32)

More information

M Sharman, K Moore, J van Leur, M Aftab, A Verrell (2014) Viral diseases in chickpeas—impact and management. GRDC Update Papers 4 March 2014, http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Viral-diseases-in-chickpeas-impact-and-management

K Moore, A Verrell, M Aftab (2014) Reducing risk of virus disease in chickpeas through management of plant density, row spacing and stubble. GRDC Update Papers 4 March 2014, https://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Reducing-risk-of-virus-disease-in-chickpeas

Watch for chickpea viruses near canola (2013)

More information

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/02/Developing-a-plan-for-chickpeas-2013

[10] M Sharman, K Moore, J van Leur, M Aftab, A Verrell (2014) Viral diseases in chickpeas—impact and management. GRDC Update Papers 4 March 2014, http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/03/Viral-diseases-in-chickpeas-impact-and-management.

[11] R Bowman (2013) Watch for chickpea viruses near canola. GRDC Media Centre May 2013, https://grdc.com.au/Media-Centre/Media-News/North/2013/05/Watch-for-chickpea-viruses-near-canola

Other less common viruses:

- Lettuce necrotic yellows virus (LNYV)
- Clover yellow vein virus (ClYVV)

9.11.4 Symptoms

Luteoviruses will kill plants within 3–4 weeks of symptoms showing.

The diseased plants have a scattered distribution, usually occurring around the edges of a crop or in areas where plant numbers are low.

In Desi varieties of chickpeas, the leaves and stems become red or brown, whereas Kabuli varieties turn yellow. In older plants that are podding, premature death may be the only obvious symptom.

A shallow cut with a knife at the base of the main stem often reveals that the stem has turned brown, compared with a white or green colour in healthy plants.

Figure 32: Symptoms of Cucumber mosaic virus include reddening of the leaves and stunted growth. Symptoms are often confused with nutritional deficiency or herbicide damage. (Photo: G. Cumming, Pulse Australia)

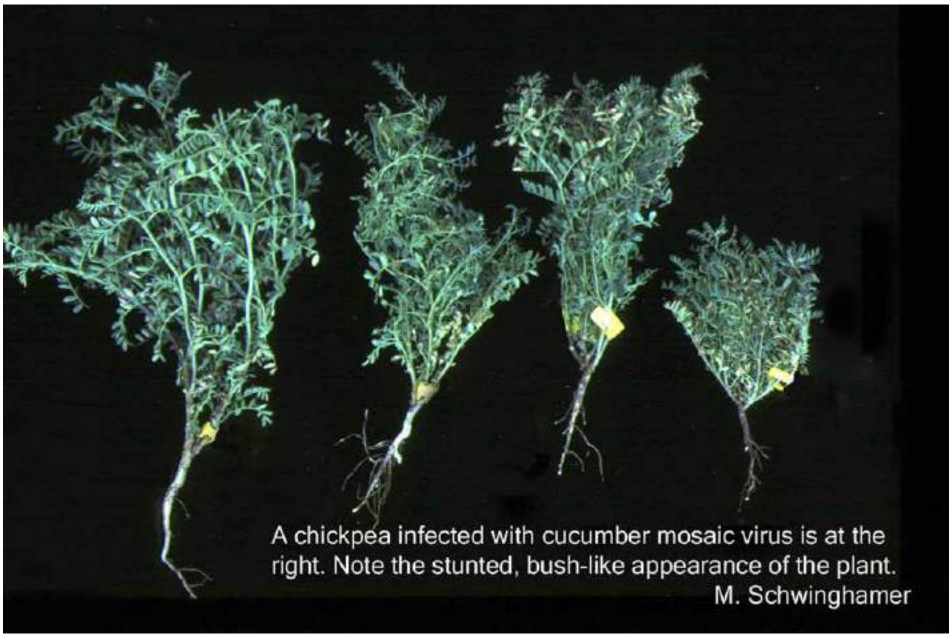

Figure 33: Shoot-tip virus complex. (Photo: M. Schwinghamer, NSW DPI)

The symptoms of AMV and some other viruses are similar to Luteovirus but more pronounced on the shoot tops. The symptoms include a pale colouration, bunching with small leaves, tip death, and the shoots are horizontal or even pointing downwards.

9.11.5 Chickpea virus-testing resource

As part of a new GRDC project, NSW DPI researcher Dr Jenny Wood is working on eliminating grain defects in Desi and Kabuli chickpeas (Figure 34). The information will assist chickpea breeders to breed for tolerance to seed markings in future varieties.

More information

[M Schwinghamer, T Knights, K Moore (2009). Virus control in chickpea - special considerations]

The two defects being examined are:

- seed markings (particularly seeds with tiger stripe or blotch markings); and
- weather-damaged seed (symptoms include light weight, brittle seeds or sprouting).

Samples are requested, whether they look clean or contain visibly diseased, marked or weathered seeds.

The grain will also be tested for germination, emergence, seed-borne diseases and moulds, by Dr Kevin Moore, Northern NSW Integrated Disease Management, NSW DPI, and results returned.

Please send a sample of harvested grain, ideally 1 kg, secured in two plastic bags (double-bagged). Do not hand pick the sample, as it must be representative of your entire harvested crop.

Send with the sample testing form, which includes:

- variety
- address of the crop paddock (GPS coordinates if possible)
- dates the crops was sown and harvested (plus flowering and maturity dates if you have them)
- information on any stressors the crops suffered in the field, including moisture or heat stress
- observations of any reddening of the foliage
- details of other varieties nearby that were affected

Samples should be addressed to:
Dr Jenny Wood
C/- Kate Keir
Tamworth Agricultural Institute
4 Marsden Park Rd
Calala NSW 2340

Figure 34: NSW DPI is now testing seed for germination, emergence, seed-borne diseases and moulds, as well as examining seed defects as part of a GRDC-funded project.

9.12 Fungal disease control

9.12.1 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.

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.

9.12.2 Principles of spraying

A fungicide spray at the commencement of flowering protects early podset. 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 about 10 days.

Timing of fungicide sprays is critical (Table 6). 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.

Table 6: Principles of when to spray for fungal disease control in chickpea

Disease	Occurrence	When to spray
Ascochyta blight	First appears under wet conditions	Ascochyta blight is spread by rainfall. 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)

Table 7 illustrates the relative importance of various forms of carryover of fungal disease infection for chickpeas and other pulses.

Table 7: Carryover of major pulse diseases, showing their relative importance as sources of infection

Disease	Stubble	Seed	Soil
Ascochyta blight	★★★	★★	★
Botrytis grey mould	★★★	★★★	★
Phytopthora root rot			★★★
Sclerotinia rot	★	★	★★★

9.13 Registered fungicides

Table 8 provides a list of registered seed dressings for chickpeas.

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

Prior to the use of any crop protection product, ensure that is currently registered or that a current permit exists for its use in chickpeas.

Registered labels and current permits can be found on the APVMA website (www.apvma.gov.au).

Table 8: Seed dressings registered for use with chickpea

✓, Registered product label claim

Active ingredient	Thiram	Thiram + thiabendazole	Metalaxyl-M
Example trade name	Thiraflo®	P-Pickel® T	Apron® XL
Ascochyta blight	✓	✓	
Botrytis grey mould	✓	✓	
Damping-off (Kabuli)	✓	✓	✓
Phytopthora root rot			✓
Jurisdiction	All States	All States	Qld, NSW, Vic, SA, WA

Table 9 provides a summary of the main disorders affecting chickpea, their causes, transmission, symptoms and management.

Table 9: Key features of the main chickpea disorders

Disorder and cause	Seed-borne?	Symptoms	Distribution and occurrence	Survival and spread	Management
Seed-borne root rot: *Botrytis cinerea* *Ascochyta rabiei* (very rare)	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; higher temperatures, i.e. later in season	Caused by insufficient supply of oxygen to roots	Avoid low-lying or poorly drained paddocks or areas within paddocks

Disorder and cause	Seed-borne?	Symptoms	Distribution and occurrence	Survival and spread	Management
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 1–5m 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
AMV (*Alfalfa mosaic virus*), CMV (*Cucumber mosaic virus)*	Yes	Initially bunching, reddening, yellowing, wilting or death of shoot tips; later discoloration.	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)	?

Source: K. Moore, NSW DPI and M. Fuhlbohm, Qld Gov.

SECTION 10

Plant growth regulators and canopy management

Not applicable for this crop.

SECTION 11

Crop desiccation and spray out

More information

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/02/Developing-a-plan-for-chickpeas-2013

http://www.daf.qld.gov.au/plants/field-crops-and-pastures/broadacre-field-crops/chickpeas/harvesting-and-storage

Pulses can be desiccated pre-harvest to enable earlier harvest and to dry out green weeds. It is becoming common practice, particularly in chickpeas, field peas and lentils. Timing is based on crop stage, and is similar to, or later than, that for windrowing.

The danger of premature desiccation lies in having excessive green cotyledons in the sample, staining of the seed coat or small seed all of which create marketability problems.

In chickpeas, desiccation can occur when fewer than 20% of pods are green and 90% of seed is changing from a green colour.

Desiccation is a valuable management tool especially under the following conditions:

- If there is a problem with green weeds at harvest.
- For improved harvest efficiency. Desiccation eliminates many of the problems associated with green stems and gum build-up, which cause uneven flow of material through the header, and 'jamming' problems. Minimising jamming enables drum speeds to be reduced in many cases, with a reduction in cracked or damaged grain.
- Where 'early' summer rain causes reshooting and re-flowering of chickpeas.
- Where there are problems of patchy or delayed crop maturity on heavy clay soils.
- Where 'early harvest management' is being adopted. [1]

11.1 Seed and pod development

Chickpea plants are indeterminate and the period of flowering can extend from 20 to 50 days depending on levels of flower abortion and the impact of moisture stress on the plant.

Causes of flower abortion and poor podset 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 about 2 days between successive nodes on each fruiting branch.

Under favorable conditions, the time taken from flowering to the visual appearance of the pod (podset) is about 6 days. After podset, the pod wall grows rapidly for the next 10–15 days to assume full pod size.

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

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. [2]

11.2 Timing of desiccation

The optimal stage to desiccate the crop is when the vast majority of seeds have reached physiological maturity (i.e. 90–95% of the crop).

The best guide at present is to base this on a visual inspection of the seeds in the top 25% of uppermost pods on each main fruiting branch (Figure 1).

Seeds are considered physiologically mature when the green seed colour begins to lighten (Figure 2).

The Western Australian recommendation of physiological maturity is 'when the pod wall begins to yellow' (Figure 3).

More investigation is required to define more clearly when physiological maturity is occurring based on a visual inspection of seeds and pods in the field.

However, until there is a more precise recommendation, the advice is to desiccate when 80–85% of pods within the crop have turned yellow–brown (Figure 4). [3]

More information

http://thebeatsheet.com.au/chickpeas/managing-helicoverpa-larvae-in-chickpea-crops-close-to-dessication-and-harvest

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

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

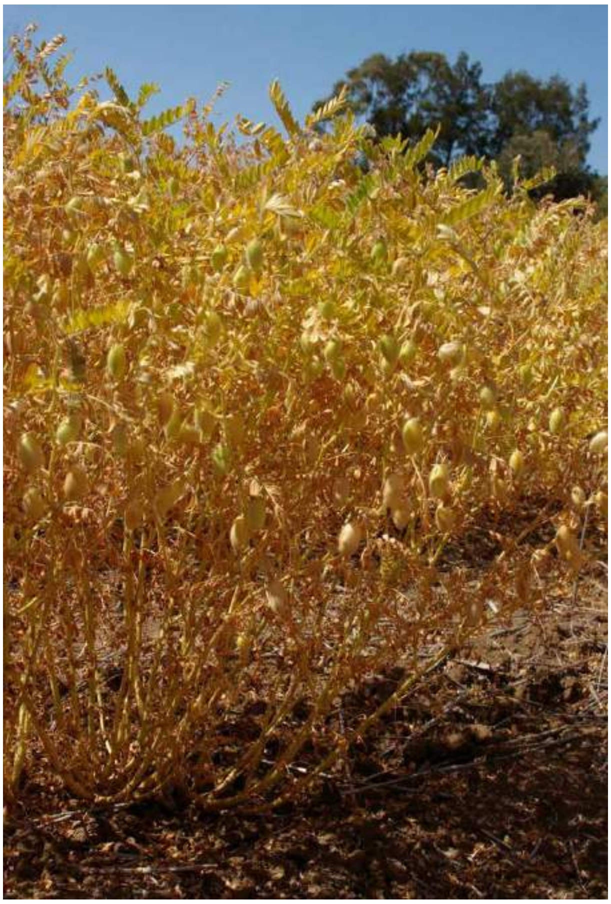

Figure 1: Correct desiccation timing based on inspection of uppermost pods of each fruiting branch.

Figure 2: Pods in the top 25% of the canopy should mainly be in the final stages of grain-fill, where the yellow colouring is moving from the 'beak' down through the seed.

Figure 3: The bottom 75% of pods should have reached maturity. Seeds have turned yellow and the pod has been bleached to a very light, green–yellow.

Figure 4: Full maturity, known as 'rattle pod', where the seed has detached from the pod wall and will rattle when shaken.

(Photos: G. Cumming, Pulse Australia)

11.3 Effect of desiccants on green immature seeds

Applying desiccants to seed that is still green and actively filling will result in:

- a reduction in grain size (and yield)
- an increase in a greenish discoloration of the seed coat
- a reduction in seed viability (dead or abnormal seed) (Table 1) [4]

[4] Pulse Australia Northern (2013) Chickpea best management practices training course manual—2013. Pulse Australia Limited.

Table 1: Effects of desiccation timing on seed viability

Treatment	Crop stage	% Normal seed	% Abnormal seed	% Dead seed
Site: Dalby				
None	Mature pods	87	9	4
Ally® & Roundup®	Mature pods	85	13	2
Roundup®	Mature pods	84	14	2
Site: Cambooya				
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).

11.4 Products registered for the desiccation of chickpea

Table 2 provides details of the products registered for use as desiccants.

Table 2: Active ingredients and trade names of chemicals used as desiccants, with critical comment extracts from the Reglone® and Roundup PowerMax® product labels [5]

Note: Always read the label supplied with the product before each use

Active ingredient	Example trade name	Rate	Critical comments
Diquat	Reglone® (200 g/L)	2–3 L/ha	Spray as soon as the crop has reached full maturity. Helps overcome slow and uneven ripening and weed problems at harvest. DO NOT harvest for 3 days after application
Glyphosate	Roundup PowerMax® (540 g/L)	0.68–1.8 L/ha	Apply when physiologically mature and <15% green pods. Use higher rates where crops or weeds are dense and where faster desiccation is required. DO NOT harvest within 7 days of application
Glyphosate plus metsulfuron	Roundup PowerMax® (540 g/L) plus Ally® herbicide (600 g/kg)	0.5–1.1 L/ha plus 5 g/ha	Apply when chickpeas are physiologically mature and less than 15% of green pods are present. Use higher rates where crops or weeds are dense and where faster desiccations is required. DO NOT harvest within 7 days of application

11.5 Windrowing

Windrowing of chickpeas is possible, but 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.

5 Pulse Australia Northern (2013) Chickpea best management practices training course manual—2013. Pulse Australia Limited.

Despite this, provided the windrows are large enough and compacted, then 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. Harvesting time can therefore be improved.

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. Use of headers in rougher country can damage knife fingers and sections, retractable fingers and other components, because of sticks and stones. 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. [6]

11.6 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 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). [7]

More information

Late season herbicide use, Fact sheet (2012)

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

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

SECTION 12

Harvest

More information

GRDC Fact Sheet 'Chickpea disease management': http://www.grdc.com.au/Resources/Factsheets/2013/05/Chickpea-disease-management

L Jenkins, K Moore, G Cumming, Pulse Australia, Chickpea: Sourcing high quality seed

'Harvesting and storage of Desi type chickpeas' (J Cassells and L Caddick): http://storedgrain.com.au/wp-content/uploads/2013/06/chickpea_harvest_storage.pdf

http://grdc.com.au/Media-Centre/Ground-Cover/Ground-Cover-Issue-89-November-December-2010/Clean-harvest-needed-to-stop-ascochyta

Chickpea harvest often coincides with wheat harvest but is considered a lower priority because of the wheat crop's potential quality premiums. However, this thinking needs to be balanced with the potentially higher value of chickpeas and potential losses from a late chickpea harvest. Chickpea yields can average about 70% of wheat yields when sown in an identical situation, in most years.

Harvest timing will depend on the moisture content that is acceptable for delivery or storage. This will be influenced by who is buying the grain, and whether aeration is available in the storage. Generally, harvest should be under way when upper pods have 15% moisture, if aiming to deliver at 13–14%.

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. [1]

Figure 1: Chickpea harvest under way near Thallon, south-west Queensland. (Photo: R. Bowman, Seedbed Media)

Diseases such as Ascochyta blight, Phytophthora root rot and root-lesion nematode 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.

[1] DAFF (2012) Chickpea—harvesting and storage. Department of Agriculture, Fisheries and Forestry Queensland, http://www.daf.qld.gov.au/plants/field-crops-and-pastures/broadacre-field-crops/chickpeas/harvesting-and-storage

If possible, clean headers and sowing equipment to remove grain, soil and stubble before moving from property to property. Spray rigs should also be cleaned to reduce the risk of disease transmission. [2]

12.1 Planning for an early harvest

Chickpeas should be harvested as soon as they mature, as pods will fall if harvest is delayed. [3] Harvesting early also minimises infection of seed. Crop desiccation enables even earlier harvest. [4]

Chickpea plants are indeterminate and the period of flowering can extend from 20 to 50 days depending on levels of flower abortion and the impact of moisture stress on the plant. Early or timely harvest of the chickpea crop has the potential to increase returns by up to 50%. Management to ensure timely harvest consists of a combination of strategies. [5]

12.2 Paddock selection and agronomy

Planning before and during sowing can reduce many harvest difficulties. Paddock selection will determine the risk of disease, waterlogging, weeds and poor establishment, ultimately influencing crop maturity. Sowing method and row spacing will affect evenness, crop height and lodging potential. All of these factors can affect the ease and timeliness of harvest. [6]

12.3 Harvest timing and technique

Chickpeas have traditionally been harvested after wheat. Costs of delaying chickpea harvest may be considerable, so need to be weighed against potential losses for wheat. Agronomists report that many growers consider losses in chickpeas will generally be less than in cereals.

Delayed chickpea harvest can have several consequences:

- Yield losses can occur from pod-drop as weathering weakens the hinge attaching the pod to the stem.
- Weathered pods become more difficult to thresh, resulting in grain loss in unthreshed pods discarded out of the back of the header, cracked grain and a slower harvest.
- Increased lodging: the risk is higher if the crop is high yielding and has been planted on wide rows.
- Harvesting at 8% moisture, instead of 13%, results in a harvest weight loss equivalent to $25/tonne (t). Farmer experience has shown yield losses of up to 30% if harvest is delayed 2–4 weeks.
- Weathered or drier grain is more likely to crack when handled, increasing the amount of split grain in the sample. The number of unthreshed pods in the sample will increase as they become harder to thresh with weathering. Both of these can result in rejection or the need for grading to meet market requirements.

2 GRDC (2013) Chickpea disease management (Southern and Northern Regions). GRDC Factsheets May 2013, http://www.grdc.com.au/Resources/Factsheets/2013/05/Chickpea-disease-management

3 Grain Legume Handbook Committee (2008) Harvesting. In ‘Grain legume handbook for the pulse industry’. Supported by the Grains Research and Development Corporation (GRDC), http://www.grdc.com.au/uploads/documents/9%20Harvesting.pdf

4 GRDC (2013) Chickpea disease management (Southern and Northern Regions). GRDC Fact Sheet May 2013, http://www.grdc.com.au/Resources/Factsheets/2013/05/Chickpea-disease-management

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

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

- The germination rate and vigour of planting seed will be reduced by weathering. Crops intended for seed are best harvested at 14–16% moisture and dried or aerated back to 12% moisture to maximise germination and vigour.
- Chickpea grain discolours and darkens with weathering, reducing its desirability, particularly in the container market.
- Chickpea prices can reach peaks during harvest to meet shipping schedules. Earlier harvesting may allow access to these opportunities.
- Darker, weathered seed may be discriminated against in the market.
- *Phoma (Ascochyta) rabiei* can infect senescing pods under wet conditions, leading to Ascochyta blight and discoloured seed.
- Late-harvested crops, particularly where there is regrowth, can be a major source of *Heliothis* (*Helicoverpa*) migration into neighbouring summer crops. [7]

12.4 Impact of delayed harvest on profitability

Early harvest of pulses is critical because delays can result in significant yield losses due to lodging, shattering and pod loss. Grain quality can also suffer. Moisture levels at harvest affect the quality of the grain in storage.

If harvesting grain for seed, germination rates are improved if grain is harvested at 12–14% and then stored in aerated silos or immediately graded and bagged. Crop-topping with herbicides prior to crop maturity may reduce grain quality and seed germination.

Harvest delays in chickpeas cost growers and the pulse industry a lot of money. In any production area, a spread of up to 4–6 weeks can occur in the harvesting of chickpea crops planted on the same sowing rain. Many of the late-harvested crops often have moisture content down to about 8%, whereas the maximum moisture content for receival is 14% and the preference is for 12%. [8]

12.4.1 Yield losses

Yield losses increase significantly the longer harvest is delayed (Figure 2).

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.

Yield losses of up to 30% have been recorded in the field. Grain losses due to a 2–4 week delay in harvest were estimated at AU$93–238/ha, depending on seasonal conditions. Most of the losses were due to pod loss at the header front, or unthreshed pods discarded out of the back of the machine.

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

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

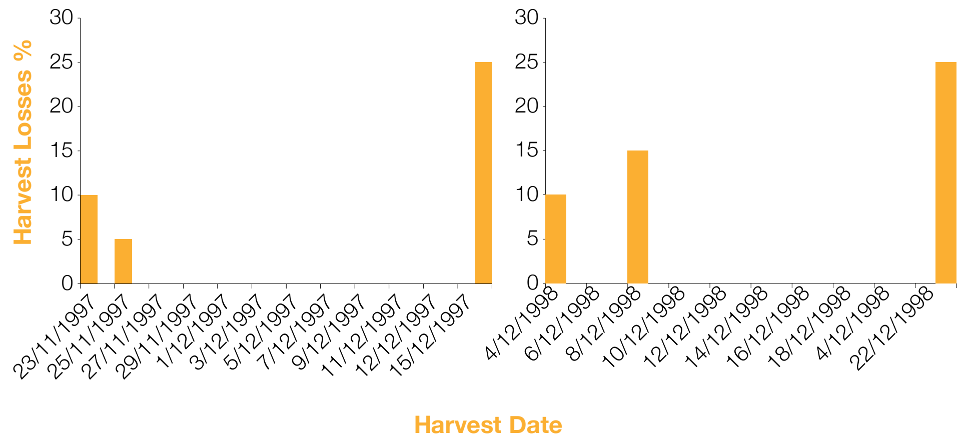

Figure 2: Harvest yield losses in 1997 (left) and 1998 (right).

Lodging can increase 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.

Loss of moisture below the Grain Trade Australia (GTA) receival standard of 14% moisture content 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. [9]

12.4.2 Deterioration in grain quality

Grain quality deteriorates the longer mature chickpeas are exposed to weathering in the field:

The chickpea seed coat is very prone to cracking if it has been exposed to wetting and drying events caused by rain or heavy dew. Expansion of the seed as it absorbs moisture, followed by contraction as it dries, weakens the seed coat, rendering it much more susceptible to mechanical damage during harvest and handling operations.

Levels of cracked and damaged grain can be as high as 50% in extreme cases of field weathering and prolonged rainfall.

Chickpeas that do not meet the Export Receival Standard of 6% maximum 'defective' chickpeas will 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.

Early-harvested chickpea seed is much more resilient to breakage during harvesting and subsequent handling, even at low moisture contents.

Desi chickpeas are ultimately processed into dhal or flour by removing the seed coat (hull) and splitting the cotyledons. The process uses abrasive-type mills to gradually abrade the seed coat from the cotyledons, and is reliant on the seed coat being firmly attached to the cotyledons.

Cracking and weakening of the seed coat prior to processing substantially reduces the recovery percentage of dhal, as well as reducing the quality of the final product.

Field-weathered chickpeas after rain are also more difficult to thresh out at harvest, and often contain much higher levels of unthreshed pods and pod material. [10]

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

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

12.4.3 Chickpea seed discoloration

Chickpea seeds discolour and darken when exposed to field weathering.

Darkening of the seed coat is caused by oxidation of polyphenol compounds. The following conditions play a major role in accelerating seed coat darkening:

- rainfall
- cool-mild temperatures
- high humidity

Figure 3: Pistol chickpea showing tiger stripe. (Photo: Jenny Wood, NSW DPI)

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 (Canada, Turkey, 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.

Weathering of seed due to delays in harvesting can substantially increase mould infection levels. 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

More information

http://www.grdc.com.au/Research-and-Development/Major-Initiatives/PBA/~/media/76F25DBABAD348EB87A12682E557CBA5.ashx

GrainCorp, Chickpea standards 2015-2016

Australian Pulse Standards 2014/2015, Pulse Australia

of fungi in late-harvested chickpea crops. While *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 develop on dry senescing pods under wet conditions, and can penetrate through to the seed. The current Export Receival Standard for visible Ascochyta blight lesions is a maximum of 1% on the seed cotyledon (kernel).

For the current Australian Pulse Standards, go to http://www.pulseaus.com.au/marketing/receival-trading-standards.

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 chickpeas, and they cannot exceed the tolerance level of 6%.

12.4.4 Missed marketing opportunities

Delayed harvest can often mean that growers miss out on premiums that are paid for early-harvested crops. This is the case in many years, with the possible exception being where major production problems have been encountered and there is a shortage in the market place. Premiums of $50–100/t for the earlier harvested crops have been paid in some years.

Early harvest gives the grower some control over how and when the crop is marketed, whereas late-harvested chickpeas can be 'price-takers' in a falling market. [11]

12.5 Implementing early harvest management

A range of management components contribute to an early crop. They can all be important at different times and for different reasons. It is important to understand the potential and limitations of each management component. 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.

These components include:

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.

2. In-crop management

- Control Botrytis grey mould if present during flowering.
- Control native budworm during flowering to maximise early pod set.
- Avoid using herbicides that delay crop maturity, such as flumetsulam (e.g. Broadstrike®).

3. Harvest management

- Consider using Roundup Power MAX® + Ally® (or equivalent registered products) to terminate the crop at 80–90% yellow–brown pod stage.
- Set the header up to operate efficiently at 14–15% grain moisture content.
- A major advantage of high-moisture harvesting is that harvest can commence earlier in the season and earlier each day.

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

- Harvesting at 14% moisture content, 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% moisture. [12]

12.6 Harvesting and header settings

Pulses are easily threshed, so concave clearances should be opened and the drum speed reduced.

If there are many summer weeds, the drum speed may have to be increased to ensure that weeds do not block the machine. Pulse grains are larger than wheat, so a concave with many wires or blanked-off sections can stop grain separation. To get the best performance, alternate wires and blanking-off plates will have to be removed. Maximum wind settings and barley sieves should ensure a good sample.

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.

When harvesting pulses for seed, take extra care to reduce grain cracking, even if this means making a poor sample. Gentle harvesting will give the best seed quality. Rotary harvesters are gentler on the crop and will generally cause less grain damage than conventional harvesters.

Chickpeas can be harvested with minor adjustments and modifications to equipment. Open-front or pick-up fronts are best suited to the job.

Chickpeas should be harvested as soon as they mature, as pods will fall if harvest is delayed.

The crop varies in height from 15 to 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 (400–600 rpm) and concave (10–30 mm) to suit (Table 1). Removing alternate wires and blank-off plates from the concave will help reduce cracking. If possible, cover the rasp bars with plate.

Video

Pulse Australia video: http://www.youtube.com/watch?v=oUBym9wa5wY

Harvesting grain at high moisture levels up to 14% should minimise cracking.

Early harvesting, before summer weeds become a problem, will reduce clogging and sample contamination. Desiccating the crop will kill summer weeds and ensure even crop-ripening.

Because chickpeas are destined for human consumption, a good sample off the header is usually required. [13]

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

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

Table 1: Harvester settings for pulses

	Chickpea	Faba bean	Green lentil	Red lentil	Lupin	Pea	Vetch
Reel speed	Medium	Slow	Slow	Slow	Slow	Medium	Slow
Spiral clearance	High	High	Low	Low	High	Standard	Low
Thresher speed	400–600 rpm	400–600 rpm	350–450 rpm	350–450 rpm	400–600 rpm	400–600 rpm	400–600 rpm
Concave clearance	10–30 mm	15–35 mm	20–30 mm	10–20 mm	10–30 mm	10–30 mm	10–30 mm
Fan speed	High	High	High	High	High	High	Medium
Top sieve	32 mm	32–38 mm	32 mm	16 mm	32 mm	25 mm	25 mm
Bottom sieve	16 mm	16–19 mm	8–16 mm	3–10 mm	16 mm	16 mm	10–16 mm
Rotor speed[A]	700–900 rpm	700–900 rpm	350–450 rpm	350–450 rpm	700–900 rpm	700–900 rpm	Slow

Source: Grain Legume Handbook, http://www.grdc.com.au/uploads/documents/9%20Harvesting.pdf

[A]Rotary machines only.

12.7 Modifications and harvest aids

Early harvesting can solve many problems. Losses are reduced because the pods are less prone to shatter or drop. The crop is also easier to gather because it stands more erect, allowing the harvester front to operate at a greater height, reducing the soil, rock and sticks entering the harvester.

Early harvesting also means there are fewer summer weeds to clog the harvester.

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.

Grain loss can be reduced by harvesting in high humidity, at night if necessary, to minimise pod shattering. Avoid reaping in extreme heat.

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 4).

Figure 4: Finger guard. (Source: Grain Legume Handbook, http://www.grdc.com.au/uploads/documents/9%20Harvesting.pdf)

A lupin breaker is a cheap and simple device that can increase harvesting capacity to reduce grain loss. A small, serrated plate attaches to the front spiral and creates an aggressive, positive feed action to clear-cut material from the front of the knife. [14]

Air fronts help to reduce shattering losses, and minimise the amount of soil and other debris (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. [15]

Options to improve harvesting include:

1. Aussie-Air. Directs an air blast through reel fingers, and is suitable for both heavy and light crops. The manufacturer claims that an extra 15 hp is required to drive an Aussie-Air but there is also less horsepower requirement because of wider concave clearances. The actual horsepower required should be no more than for a heavy cereal crop.
2. Harvestaire. Replaces reel with a manifold that directs a blast of air into the front. The manifold causes some interference with the incoming crop. Correct orientation of air blast is important; an optional secondary fan to increase the air blast is worthwhile. The device is more effective in light crops.
3. 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.
4. Extension fingers (Figure 5). Plastic extension fingers about 30 cm long that fit over existing fingers can save significant losses, for little financial outlay, at the knife. Pods that would have fallen in front of the knife are caught on the fingers and pushed into the comb by the incoming crop.
5. Extended fronts. 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.
6. Platform sweeps. 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.
7. Draper fronts. Draper fronts such as MacDon® and Honeybee® have large clearances behind the knife and carry the crop to the elevator. The front can also be used for cereals without modification.

Note that cost benefits must be assessed; a small area of pulses may not justify the cost of some of the above modifications. [16]

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

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

16 Grain Legume Handbook Committee (2008) 'Grain legume handbook.' Supported by the Grains Research and Development Corporation (GRDC).

Figure 5: Plastic extension fingers fitted to a Draper front. (Photos: G. Cumming, Pulse Australia)

12.8 Achieving a clean sample

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.

12.8.1 Perforated screens

Perforated screens fitted on the bottom of the broad elevator, cross augers, 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.

12.8.2 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.

12.8.3 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 peas 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.

12.8.4 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. This 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.

12.8.5 Flexible cutter-bar fronts (flexi-fronts)

The cutter-bars of these 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. [17]

12.9 Fire safety

Fires can be a major hazard, as chickpea dust has a relatively low flash point. The problem is most prevalent where there has been no rain in the 2-4 weeks leading up to harvest. Risk can be minimised:

- Clean down headers regularly during the chickpea harvest.
- Dust settling on the manifold or turbo is usually the initial cause of most fires.
- Dust build-up on the header is often worse when using an air-front.
- Be wary of slipping belts and collapsed bearings that could ignite the dust.
- Keep a water tank nearby during harvest and carry a knapsack that works. [18]

12.10 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. [19]

12.11 Harvest weed-seed management in the northern grains region

A survey across 1400 transects in 70 paddocks assessed the weed distribution, density and seed production at harvest in wheat, chickpea and sorghum crops in four cropping zones of the northern grain region. Seventy weed species were identified, of which 12 were found in 7–45 paddocks. The survey identified value in investigating harvest weed-seed management options, including the Harrington Seed Destructor (HSD), to greatly reduce seed-bank replenishment of problem weeds.

Background

The 2011–12 survey was a joint effort between Department of Agriculture, Fisheries and Forestry Queensland (DAFF), Queensland Alliance for Agriculture and Food Innovation (QAAFI) and Australian Herbicide Resistance Initiative (AHRI). The focus of the project was to identify the potential for harvest weed-seed management in the northern grain region of northern New South Wales, and southern and central Queensland.

Currently, harvest weed-seed management is not practised as a weed-control option in the northern cropping regions. The potential for this approach has not been evaluated

[17] Grain Legume Handbook Committee (2008) 'Grain legume handbook.' Supported by the Grains Research and Development Corporation (GRDC).

[18] Northern Chickpea Best Management Practices Training Course Manual – 2013, Pulse Australia Limited

[19] Northern Chickpea Best Management Practices Training Course Manual – 2013, Pulse Australia Limited

in the summer or winter cropping systems across these regions. The survey was conducted to address this situation.

The approach was to identify weed species with upright, seed-bearing plant parts that could be collected during harvest of the dominant crops of these regions. The survey provides a comprehensive set of data allowing accurate determination of the potential for successful use of at-harvest, weed-seed management systems for the northern region.

Approach

A random survey was conducted on 70 paddocks of wheat, chickpea and sorghum in the four main cropping zones of the northern grain region (Table 2).

Within each paddock, 20 transects of 10 m^2 (1 m by 10 m) were selected, using a zigzag pattern to be representative of weed infestations across the paddock (this is the same protocol as used in previously published, northern region weed surveys).

The following measurements were made in each transect:

- weed species present
- density of weed species, using the rating scale (plants/10 m^2): 1, 1–9; 2, 10–49; 2.5, 50–100; and 3, >100
- visual estimation of percentage of each species seeding

For each species seeding, three representative samples were collected from each paddock and the following measurements made:

- visual estimation of percentage of seeds or seed heads above potential harvest height (nominated as 5 cm for chickpea, 15 cm for wheat, 30 cm for sorghum)
- visual estimation of percentage total seed retained at time of sampling
- number of seeds or seed heads (and no. of seeds in five representative seed heads) per plant above harvest height
- total seed production, number of seed retained, and potential for harvest management (rated as a percentage)

Table 2: Extent of northern region weed seed at harvest survey

Region and crop	Number of paddocks	Number of species present at harvest	Number of species retaining seed at harvest
Central Highlands, QLD			
Chickpea	5	8	6
Wheat	5	5	4
Sorghum	10	12	11
Darling Downs, QLD			
Chickpea	5	11	7
Wheat	5	12	10
Sorghum	10	15	11
South-west Down, QLD			
Chickpea	5	15	11
Wheat	5	8	3
Sorghum	10	25	19
Liverpool Plains, NSW			
Chickpea	5	22	16
Wheat	5	18	12
Sorghum	-	-	-
TOTAL	**70**	**70**	

Results

The weed flora was diverse, with 70 species found. There were 37 species in chickpea crops, 33 in wheat, and 38 in sorghum (Table 2). Fifteen species were found in both winter and summer crops. Of these, 70% had seed retained at harvest time.

Twelve weed species were commonly found across the cropping zones and crops in 7–45 paddocks (Table 3). The most prevalent were the weeds with wind-blown seed—sowthistle and fleabane. There were three common grasses—barnyard grass, wild oat and feathertop Rhodes grass; three brassicas—turnip weed, mustard and African turnip weed; plus five other broadleaf weeds—bladder ketmia, pigweed, native jute, Australian bindweed and wild gooseberry. Caustic weed was also present in 10 paddocks but was not seeding.

Table 3: The most common weed species seeding at harvest time in wheat, chickpea and sorghum, and data on seed loss, seed remaining and percentage of remaining seed above potential harvest height (averaged across each of four cropping zones) for each species

Seed data for each species are listed in the order wheat, chickpea and sorghum

Weed	Scientific name	Number of paddocks infested	Number of paddocks seeding	Seeds dropped per plant	Seeds remaining per plant	% above harvest height
Sowthistle	*Sonchus oleraceus*	45	38	150-10,150 2010-18,680 1290-3750	770-2040 4470-14,660 1070-8690	80-100 100 65-85
Fleabane	*Conyza bonariensis*	28	17	0-3180 0-14,230 30,210-130,060	4885-13,950 17,790-46,255 28,710-33,430	40-100 90-100 55-60
Barnyard grass	*Echinochloa spp*	20	17	200 0 3250-4350	3585 2865 730-14,040	100 60 20-25
Bladder ketmia	*Hibiscus trionum*	19	15	10 - 55-325	45 - 175-215	25 0 30-100
Wild oat	*Avena spp*	14	13	55-195 8-24 -	155-295 180-220 -	100 100 -
Turnip weed	*Raphanus raphanistrum*	10	9	- 0 25	- 150-28,170 455	0 95-100 20
African turnip weed / mustard	*Sisymbrium spp*	9	8	0 0 -	995 33,130-112,075 -	100 100 -
Pigweed	*Portulaca oleracea*	8	3			0 0 0
Native jute	*Corchorus capsularis*	8	6			0 0 0
Australian bindweed	*Convolvulus erubescens*	7	3	0	320	0 0-80 0

Weed	Scientific name	Number of paddocks infested	Number of paddocks seeding	Seeds dropped per plant	Seeds remaining per plant	% above harvest height
Feathertop Rhodes grass	*Chloris virgata*	7	7	-	-	0
				370-9905	2485-11,610	100
				0-21,940	13,640-31,040	60-75
Wild gooseberry	*Physalis minima*	7	5	-	-	-
				-	-	-
				210	11,625	15

For sowthistle and fleabane, many seeds had already dropped from the plants, particularly for sowthistle in chickpea and fleabane in sorghum. However, many seeds remained on the plants, 770–14,660 seeds/plant for sowthistle and 4885–46,255 seeds/plant for fleabane (Table 3), most of which were above the potential harvest height. Thus, these weeds are a priority for harvest weed-seed management.

Barnyard grass was the third most prevalent weed with a substantial number of seeds remaining in all three crops, although there were more seeds dropped in sorghum (3520–4350 seeds/plant) than in winter crops (0–200). A substantial proportion of feathertop Rhodes grass seeds had dropped in chickpea (370–9905) and sorghum (0–21,940), although large numbers remained on the plant above harvest height.

Several hundred seeds remained on wild oat in wheat (155–294 seeds/plant) and chickpea (180–220) but a large proportion of wild oat seed had already dropped in wheat paddocks.

The brassica weeds produced large numbers of seeds in chickpea (150–112,075 seeds/plant) but many fewer in wheat (0–995). Most seeds were above the potential harvest height, and thus these weeds are a priority for harvest weed-seed management.

Bladder ketmia, pigweed, native jute, Australian bindweed and wild gooseberry had either no seed above harvest height or small numbers of seeds, except for wild gooseberry in sorghum, with 11,625 seeds remaining.

Some less common weeds identified with large numbers of seeds per plant (in parentheses) above potential harvest height were:

- cudweed (2500–22,645)
- climbing buckwheat/bindweed (1400–9420)
- dock (30,060)
- mallow (6765)
- malvastrum (1115)
- Mexican poppy (15,970)
- New Zealand spinach (1125)
- paradoxa grass (1040)
- sida (1725)
- St Barnaby's thistle (11,045)
- stink grass (18,995)
- sweet summer grass (1660)
- wild sunflower (2750)
- windmill grass (6225)
- wireweed (820-4000)

Annual ryegrass and barley grass were found in only one paddock in the Liverpool Plains region.

Implications

This survey has shown a clear and urgent need for growers to manage weeds better to prevent large annual replenishments of the seed-bank. A potential tactic is to use one of the

More information

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2012/04/What-percent-of-northern-weed-seed-might-it-be-possible-to-capture-and-remove-at-harvest-time-A-scoping-study

Harvest weed seed control

harvest weed-seed management options, such as the HSD. It is also the ideal window for northern growers to experiment with narrow windrow burning, which is emerging as a cost-effective weed-control option.

These tactics could be used to greatly improve management of many weeds, particularly the summer and winter grasses, brassica weeds, some climbing weeds, and possibly sowthistle and fleabane if the technique is capable of capturing and destroying wind-blown seeds. [20]

[20] S Walker, M Widderick. Weed seed management at harvest. Northern Grower Alliance, http://www.nga.org.au/module/documents/download/146

SECTION 13
Storage

More information

[Australian pulse standards 2015/2016]

More information

[See the GRDC Stored Grain GrowNote and fact sheets]

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 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. [1] Effective management of stored chickpeas can eliminate all these risks to pulse quality.

Growers contemplating medium–long-term storage (6–12 months) need to be aware that chickpeas continue to age, and that quality deteriorates 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

Condition of the seed at harvest

- Seed subject to field weathering prior to harvest will deteriorate a lot quicker in storage, even when stored under 'acceptable' conditions of 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 darkening of seed, any grain stored >12% MC will require cooling.
- Growers should avoid even short–medium storage of weather-damaged grain. [2]

[1] Australian pulse standards 2015/2016. http://www.pulseaus.com.au/storage/app/media/markets/2015-16_pulse-standards.pdf

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

Figure 1: Meticulous hygiene and aeration cooling to manage storage temperature and moisture are crucial to prevent insect damage and moulds in stored chickpeas.

13.1 How to store chickpeas on-farm

Aeration of stored pulses in silos is the key non-chemical tool used to minimise the risk of insect infestations and spoiling through heat and/or moisture damage. For storage period longer than 2 months, silos with aeration cooling that can be sealed gas-tight when fumigation is required are essential (Table 1).

Well-designed and properly operated on-farm storage provides the best insurance that a grower can have to manage the quality of chickpeas to be out-turned. Storages must be used in conjunction with sound practices, which include monthly sieving for insects, regular grain quality inspections and ensuring that aeration cooling equipment is operating as required (Table 2).

Successful storage of pulses requires a balance between ideal harvest and storage conditions. Harvesting at 14% MC captures grain quality and reduces mechanical damage to the seed, but requires careful management in aerated silos to avoid deterioration during storage. [3]

3 P Burrill, P Botta, C Newman, C Warrick (2014), GRDC Fact Sheet, July 2014. https://grdc.com.au/Resources/Factsheets/2014/07/Grain-Storage-Fact-Sheet-Storing-Pulses

Table 1: Maximum recommended storage period for pulses

Moisture content	Grain temperature (°C)	
	20	30
14	3 months	N/A
13	9 months	3 months
12	> 9 months	9 months

Source: CSIRO Stored Grains Research Laboratory

Table 2: Storage life of chickpeas

Moisture content	Grain temperature (°C)			
	20	30	40	
12	66.6	16.6-21.6	3.6-4.3	Longevity of seed (months)
15	23-28	6-10	1-1.6	

More information

http://storedgrain.com.au/wp-content/uploads/2013/07/GRDC-GS-FACILITIES-Booklet-2013_Final.pdf

P Burrill, P Botta, C Newman, C Warrick (2014), GRDC Fact Sheet, July 2014

Because chickpeas are susceptible to splitting at the ideal storage MC of ≤12%, cone-based rather than flat-based silos are recommended for easy out-loading with minimal seed damage. 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 could cause it to collapse. [4]

Detailed information about selecting, locating and fitting-out silos is contained in the GRDC Grains Industry Guide: 'Grain storage facilities: Planning for efficiency and quality'.

Use of a belt conveyor instead of an auger is advisable when handling chickpeas. If movement via auger cannot be avoided, minimise the number of times that augers shift grain and adjust auger settings to ensure the chickpeas are handled as gently as possible. Follow these rules to minimise auger damage to chickpeas:

- Ensure augers are full of grain and operated at slow speeds.
- Check auger flight clearance—optimum clearance between flight and tube, in order to minimise lodging and damage, should be half the grain size.
- Operate augers as close as possible to their optimal efficiency, usually an angle of 30°.

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. This resistance has come about because of the prevalence of silos that are poorly sealed or unsealed during fumigation. [5]

The Kondinin Group 2009 National Agricultural survey revealed that 85% of respondents had used phosphine at least once during the previous 5 years, and of those users, 37% used phosphine every year for the past 5 years. A Grains Research and Development Corporation survey during 2010 revealed that only 36% of growers using phosphine applied it correctly, in a gas-tight, sealed silo (Figure 2).

Research shows that fumigating in a storage that does not meet the industry standard 'silo pressure test' does not achieve a high enough concentration of fumigant for a long enough period to kill pests at all life-cycle stages (Figure 3). For effective phosphine fumigation, a minimum of 300 parts per million (ppm) gas concentration for 7 days or 200 ppm for 10 days is required. Fumigation trials in silos with small leaks demonstrated

4 P Burrill, P Botta, C Newman, C Warrick (2014), GRDC Fact Sheet, July 2014. https://grdc.com.au/Resources/Factsheets/2014/07/Grain-Storage-Fact-Sheet-Storing-Pulses

5 C Warrick (2012) Fumigating with phosphine, other fumigants and controlled atmospheres: Do it right—do it once. GRDC Grains Industry Guide, January 2011. Reprinted Aug. 2012, http://www.grdc.com.au/~/media/5EC5D830E7BF4976AD591D2C03797906.pdf

that phosphine levels are as low as 3 ppm close to the leaks. The rest of the silo also suffers from reduced gas levels. [6]

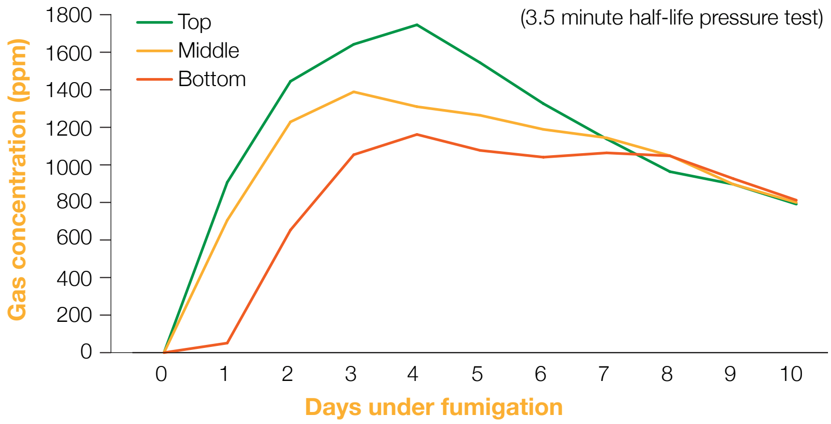

Figure 2: Gas concentration in gas-tight silo.

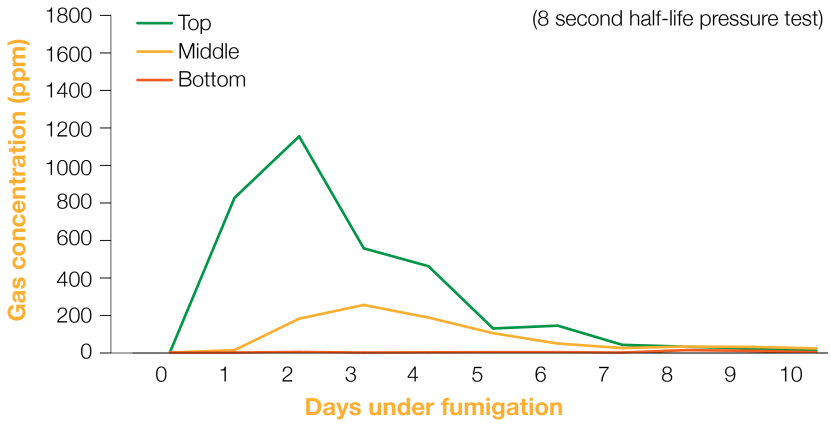

Figure 3: Gas concentration in a non-gas-tight silo.

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: http://www.youtube.com/watch?v=iS3tUbJZl6U.

To find out more about how to pressure-test silos, visit 'Fumigating with phosphine, other fumigants and controlled atmospheres' at http://www.grdc.com.au/~/media/FC440FBD7AE14140A08DAA3F2962E501.pdf.

Aeration controllers help to reduce the number of mistakes made by leaving the fan running at times of unsuitable ambient air (e.g. high relative humidity). They also reduce the time needed by operators to turn fans on and off. It remains vital that controllers and storages are checked regularly. Most controllers have hour meters fitted so run times can be checked to ensure they are within range of the expected total average of about 100 h per month.

Serious grain damage has occurred when fan performance has not met required airflow rate, as measured in litres per second per tonne (L/s.t). When cooling or drying grain

More information

https://grdc.com.au/~/media/ReFocus-media-library/Document/GRDC-Document-Store/Publications-Media-and-Communications/Factsheets/Grain-Storage-FS-Pressure-testing-sealable-silos.pdf

[6] P Botta, P Burrill, C Newman (2010) Pressure testing sealable silos. GRDC Fact Sheet, September 2010, https://grdc.com.au/~/media/ReFocus-media-library/Document/GRDC-Document-Store/Publications-Media-and-Communications/Factsheets/Grain-Storage-FS-Pressure-testing-sealable-silos.pdf

with elevated moisture levels, inadequate airflow rate and/or poor system design can see sections of the storage develop very high grain temperatures. During aeration drying, moisture drying-fronts can be moving too slowly to prevent grain spoilage. Grain quality losses from moulds and heat damage can occur rapidly. This type of damage often makes the grain difficult to sell, and in some cases may cause physical damage to the silo itself. [7]

Researchers in Australia have developed a device that measures working airflow rates of fans fitted to grain storage. Called the A-flow, it has been validated under controlled conditions using an Australian Standard fan-performance test rig to be within 2.6% of the true fan output. The device was used on a typical grain storage that was in the process of aerating recently harvested grain. A fan advertised to provide 1000 L/s (equal to 6.7L/s.t on a full 150-t silo) was demonstrated to be only producing 1.8 L/s.t. Because of this test, the farmer recognised a need to make changes to the current aeration system design.

A number of changes may be required if airflow rates are not suitable for efficient aeration cooling or drying. A new fan that is better suited to the task could be installed; a second fan added; or the amount of grain in the silo reduced to increase flow rate per tonne of grain.

A GRDC factsheet explaining how to build and use an A-flow is available at: http://www.grdc.com.au/Resources/Factsheets/2012/08/Grain-Storage-Performance-testing-aeration-systems.

Chickpeas can be stored successfully in silo bags for up to 3 months, but this 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. [8] Black discoloration of chickpeas due to moisture ingress into the base of grain bags has also occurred, causing serious losses in storage.

Insect pests in storage

Insects are not considered a major problem in stored chickpeas.

Exceptions appear to be in cases where chickpeas have higher levels of splits and damaged seed, or have been loaded into storages containing residues of cereal grain already infested with:

- *Tribolium castaneum* (rust-red flour beetle)
- *Rhyzopertha dominica* (lesser grain borer)
- *Oryzaephilus surinamensis* (saw-toothed grain beetle)

Where a prior infestation exists in storage facilities, it can spread and develop in the chickpeas. One example is where mungbeans are infested with bruchids (*Callosobruchus* spp.), which can also survive and breed at slower rates in chickpeas.

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. Good hygiene, combined with aeration cooling, should prevent infestations developing.

If weather damage prior to harvest or header setting has 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.

7 P Burrill, A Ridley (2012) Performance testing aeration systems. GRDC Northern Update, Spring 2012, Issue 66, http://www.icanrural.com.au/newsletters/NL66.pdf

8 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

Chickpea gradings are attractive to storage pests. These 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.

If insects are found in stored chickpeas, the only treatment options are controlled atmospheres (CO_2, N_2), or phosphine fumigation. 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 handle treated grain before the 7–10-day exposure period plus the required airing or venting period to remove the gas.

No insecticide sprays are currently registered for use on chickpeas. Markets are particularly sensitive to insecticide residues, so any detection of residues on chickpeas could result in loss of a market, not just rejection of a contaminated delivery.

For structural treatments of silos, use an inert dust such as diatomaceous earth (DE) after a thorough cleaning of all old grain residues. [9] Pressure-hose washing out of a silo and then leaving it open to dry is also recommended, particularly if an insect infestation occurred in the previous stored grain.

13.2 Hygiene

Effective hygiene plus aeration cooling can overcome 75% of pest problems in on-farm storage. Clean out all grain and pulse residues when silos and grain-handling equipment are not in use to help minimise the establishment and build-up of pest populations. 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. Meticulous grain hygiene involves removing any old grain that can harbour pests. These pests opt for dark, sheltered areas and breed best in warm conditions.

Successful on-farm storage hygiene involves cleaning all areas where grain and pulses become trapped. Pests can survive in a tiny amount of grain, which can go on to infest any parcel of fresh grain through the machine or storage. Clean out harvesters and handling equipment thoroughly with compressed air after use.

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.

Diatomaceous earth is an amorphous silica commercially known as Dryacide® and acts by absorbing the insect's cuticle or protective waxy exterior, causing death by desiccation. If applied correctly with complete coverage in a dry environment, DE can provide up to 12 months of protection for storages and equipment. To find out more about what to use, when and how to clean equipment and storages to minimise the chance of insect infestation, download the GRDC Grain Storage Fact Sheet: Hygiene and Structural Treatment for Grain Storages (June 2013): visit www.grdc.com.au/GRDC-FS-HygieneStructuralTreatments.

More information

www.grdc.com.au/GRDC-FS-HygieneStructuralTreatments

13.3 Fumigating chickpeas

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, and successful fumigation requires a storage that can be sealed gas-tight.

While phosphine has some resistance issues, it is widely accepted as having no residue issues for grain or pulses. The grain industry has adopted a voluntary strategy to manage the build-up of phosphine resistance in pests. Its core recommendations are to

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

limit the number of conventional phosphine fumigations on undisturbed grain to three per year, and to employ a break strategy. [10]

New research has identified the gene responsible for insect resistance to phosphine. Genetic analysis of insect samples collected from south-eastern Queensland between 2008 and 2012 has allowed researchers to confirm the increasing incidence of phosphine resistance in the region. Although few resistance markers were found in insects collected 5 years ago, an average of 5% of insects collected in 2011 carried the resistance gene. Further testing with DNA markers that can detect phosphine resistance is expected to identify problem insects before resistance becomes entrenched, and help to prolong phosphine's effective life. [11]

More information

http://www.grdc.com.au/~/media/FC440FBD7AE14140A08DAA3F2962E501.pdf

Achieve effective phosphine fumigation by placing the 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. After fumigation, open top lids and ventilate grain for a minimum of 1 day with aeration fans running, or 5 days if no fans are fitted. A minimum withholding period of 2 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 to 17 days. Read label directions.

To find out more, visit 'Fumigating with phosphine, other fumigants and controlled atmospheres: Do it right—do it once. A Grains Industry Guide'.

Non-chemical treatment options 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 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 produces no residues, so grains can be traded at any time, unlike chemical fumigants that have withholding periods. Insect control with N_2 involves a process using pressure swinging adsorption (PSA) technology, modifying the atmosphere within the grain storage to remove everything except N_2, starving the pests of oxygen. [12]

Silo bags can also be fumigated. Research conducted by Andrew Ridley and Philip Burrill from Department of Agriculture Fisheries and Forestry, Queensland, with Queensland farmer Chris Cook has found that 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 about 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 (Figure 4). [13]

[10] 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/Phosphine_Strategy.pdf

[11] D Schlipalius (2013) Genetic clue to thwart phosphine resistance. GRDC Ground Cover Issue 102, Jan.–Feb. 2013, http://www.grdc.com.au/Media-Centre/Ground-Cover/Ground-Cover-Issue-102/Genetic-clue-to-thwart-phosphine-resistance

[12] C Warrick (2012) Fumigating with phosphine, other fumigants and controlled atmospheres: Do It right—do it once. A Grains Industry Guide. January 2011. Reprinted June 2012, http://www.grdc.com.au/~/media/5EC5D830E7BF4976AD591D2C03797906.pdf.

[13] P Burrill, A Ridley (2012) Silo bag fumigation. GRDC Northern Update, Spring 2012, Issue 66, http://www.icanrural.com.au/newsletters/NL66.pdf

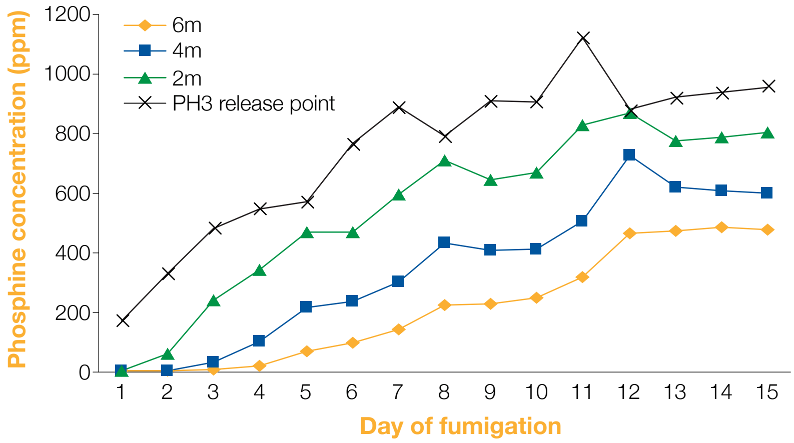

Figure 4: Spread of phosphine gas in a silo bag from a release point to gas-monitoring lines at 2, 4 and 6 m along a silo bag.

13.4 Aeration during storage

Pulses stored above 12% moisture content require aeration cooling to maintain quality. Australian Pulse Standards are set at a maximum moisture limit of 14% for most pulses, 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 lower grain temperatures, which will minimise the effects of seed darkening, declining germination and seed vigour. Aeration cooling allows for longer term storage of low-moisture grain by creating desirable conditions for pulses and undesirable conditions for mould and pests (Table 3 and Figure 5). Unlike aeration drying, aeration cooling can be achieved with airflow rates as little as 2–3 L.s.t for pulses. High-moisture pulses can also be safely held for a short time with aeration cooling before blending or drying. Fans should be run continuously to prevent self-heating and quality damage. Inspect grain often when holding any high-moisture grain.

Pulses stored for longer than 6 weeks with a high moisture content of >14% will require drying or blending to maintain seed quality. Aeration drying has a lower risk of cracking and damaging pulses, which can occur with hot-air dryers. Unlike aeration cooling, aeration drying requires high airflow rates of at least 15–25 L/s.t and careful management. For more information on aeration drying, refer to the GRDC booklet 'Aerating stored grain, cooling or drying for quality control': http://www.grdc.com.au/GRDC-Booklet-AeratingStoredGrain.

Table 3: The effect of grain temperature on insects and mould (Kondinin Group table).

Grain temperature (°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 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

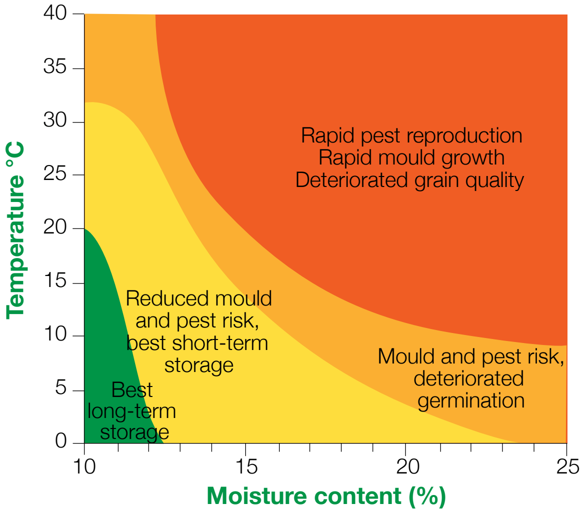

Figure 5: Effects of temperature and moisture on stored grain.

Source: CSIRO Ecosystems sciences as published in http://www.grdc.com.au/~/media/36D51B725EF44EC892BCD3C0A9F4602C.pdf

13.5 Monitoring chickpeas in storage

Like cereal grains, chickpeas need to be delivered with nil live storage insects. [14] Growers are advised to monitor all grain storages every 2 weeks during warmer periods of the year and at least monthly during cool periods of the year. It is essential that insect pests present in the on-farm storage environment are identified so that growers can exploit the best use of both chemical and non-chemical control measures to control them.

Through sieving and quality inspections, monitor your stored pulses and keep records of what you find. Use one of the GRDC stored pest identification publications. Also, record any fumigations. If safe, visually check, smell and sample grain at the bottom and top of the stack regularly. [15] Having sample ports fitted in the side of the silos also enables temperature probe checks and grain sampling.

Photographs and descriptions of pests can be found in the GRDC's 'Stored grain pest identification: Back pocket guide'. Download it from: http://www.grdc.com.au/~/media/8253D697BA6F4BF3AA5B5CBDFA7F4D2D.pdf.

This fact sheet outlines how to monitor your stored grain for infestations. 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 stores for early pest detection. Probe or pitfall traps placed into the top of the grain will often detect storage pest insects before you may 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 on to 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. The rice weevil, cowpea bruchid and saw-toothed grain beetles can walk up the walls of the glass easily, but flour beetles and

[14] Australian pulse standards 2015/2016. http://www.pulseaus.com.au/storage/app/media/markets/2015-16_pulse-standards.pdf

[15] P Burrill, P Botta, C Newman (2010) Aeration cooling for pest control, GRDC Grain Storage Fact Sheet, September 2010, http://www.grdc.com.au/~/media/AB8938CFDCCC4811AD218B45C308BEBD.pdf

More information

http://www.grdc.com.au/~/media/3BFEE576F15F4AB8A6681659BE0A1627.pdf

lesser grain borer cannot. Look closely at the insects walking up the glass. Rice weevils have a curved snout at the front, saw-toothed grain beetles do not, and the cowpea bruchid has a globular, tear-shaped body. [16]

New research in southern and central Queensland has shown that industry may need to consider an area-wide approach to pest and resistance management. The research has indicated flight dispersal by the lesser grain borer and the rust-red flour beetle, both of which are major insect pests of stored grain. The research involved setting beetle traps along a 30-km transect in the Emerald district, which showed that the lesser grain borer is flying all year round in central Queensland, whereas the flour beetle appeared to be located mainly around storages during winter and then spread out into the surrounding district in summer. This study highlights the importance of finding and dealing with infestations to limit the number of pests that can infest clean grain. [17]

13.6 Structural treatments for chickpea storages

13.6.1 Applying inert dust

Inert dust requires a moving air-stream to direct it onto the surface being treated; alternatively, it can be mixed into a slurry with water and sprayed onto surface. See label directions. Throwing dust into silos by hand will not achieve an even coverage, 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 on-farm use, connected to a venturi duster such as the Blovac BV-22 gun (Figure 6).

Figure 6: A blow/vac or air venture gun is the best applicator for inert dusts. (Photo: C. Warrick, Kondinin Group)

The application rate is calculated at 2 g/m^2 surface area treated. Although inert, breathing in excessive amounts of dust is not ideal, so use a disposable dust mask and goggles during application (Table 4).

13.6.2 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.

16 P Burrill, P Botta, C Newman, B White, C Warrick (2013) Northern and southern regions stored grain pests—identification. GRDC Grain Storage Fact Sheet June 2013, http://storedgrain.com.au/northern-southern-regions-stored-grain-pests-identification/

17 G Daglish, A Ridley (2012) Stored grain insects: how they spread and implications for resistance, GRDC Northern Update, Spring 2012, Issue 66, http://www.icanrural.com.au/newsletters/NL66.pdf

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. [18]

Table 4: Inert dust (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
1800	2.60

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

SECTION 14

Environmental issues

14.1 Frost and temperature issues for chickpeas

Effects of temperature and frost damage

The three major factors affecting chickpeas are temperature, day length and drought. In contrast to other winter legumes, chickpeas are particularly susceptible to cold conditions, especially at flowering.

Frost damage to vegetative growth

Damage is more likely to occur where the crop has rapidly grown during a period of warm weather, and is then subjected to freezing temperatures. The visible effect may occur as patches in the field, on individual plants or on branches of plants. Damage is usually more severe where stubble has been retained. Symptoms include marginal bleaching of the leaflets and a 'hockey stick' bend in branches (Figure 1). The plant could show signs of wilting and desiccation of the leaves.

Other management practices can increase the risk of frost damage, such as carry-over atrazine residues in sorghum and row orientation. Varieties can also differ markedly in their response to frost. Regrowth will generally occur where there is adequate soil moisture.

Frost damage to flowers and pods

Freezing temperatures destroy flowers and young developing seed. Pods with aborted grain lose their green colour and hay-off. Pods with peas at a later stage of development are generally more resistant and may suffer only from a mottling and/or darkening of the seed coat.

Low-temperature flower abortion

Low temperatures can also cause flower abortion. The most important aspect of temperature is not the maximum or minimum daily temperature but the average daily temperature. Where average mean daily temperatures are <15°C (max. temp + min. temp divided by 2), pollen viability is reduced and flowers will fail to develop into pods.

High-temperature flower abortion

Temperatures >35°C cause flower abortion and may result in a lowering of yield potential due to a shorter amount of time available for seed-filling. Growers must consider planting time, and weigh up the potential yield benefits of early planting against the risk of frost and cold temperatures at flowering. [1]

More information

http://www.daf.qld.gov.au/plants/field-crops-and-pastures/broadacre-field-crops/chickpeas/planting

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

Figure 1: Frost can cause bends like a hockey-stick in chickpea stems. (Photo: S. Loss, DAFWA)

Late frosts also cause flower, pod and seed abortion (Figure 2). Pods at a later stage of development are generally more resistant to frost than are flowers and small pods, but may suffer some mottled darkening of the seed coat.

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. Minimum temperatures <5°C during the reproductive stage will kill the crop, but new regrowth can occur from the base of the killed plants if moisture conditions are favourable.

Figure 2: 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)

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.

In Australia, drought stress often accompanies high temperatures in spring, causing the abortion of flowers, immature pods and developing seeds. High levels of humidity and low light also prevent pod set. [2]

More information

Journal article in *Crop & Pasture Science* by Devasirvatham et.al. (2012): High temperature tolerance in chickpea and its implications for plant improvement

http://www.dpi.nsw.gov.au/archive/agriculture-today-stories/ag-today-archives/may-2010/frosts-and-low-temps

http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0006/431268/Chickpea-time-of-sowing-trial.pdf

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

See also GRDC GrowNotes (Chickpeas) Section 3: Planting and Section 4: Plant growth and physiology.

14.2 Tolerance to low temperature

Research overseas and within Australia has demonstrated a range of cold tolerance among chickpea varieties. In parts of the world where chickpea is 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 and 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 found. Some genotypes from India are less sensitive than those currently grown in Australia, and these are being utilised in chickpea-breeding programs at Department of Agriculture and Food Western Australia (DAFWA) and the University of Western Australia (UWA).

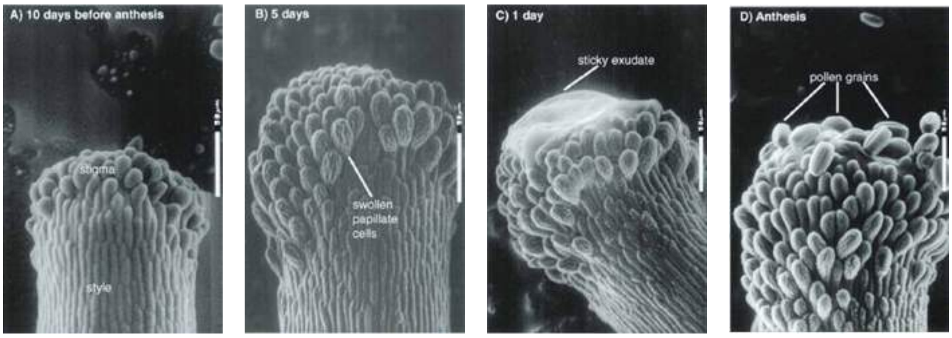

Figure 3: Development of the style and stigma of chickpea flowers taken with an electron microscope. (Photo H. Clarke, UWA)

Controlled environment studies at University of Western Australia have identified two stages of sensitivity to low temperature in chickpea. The first occurs during pollen development in the flower bud, resulting in infertile pollen even in open flowers. The second stage of sensitivity 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 (Figure 3).

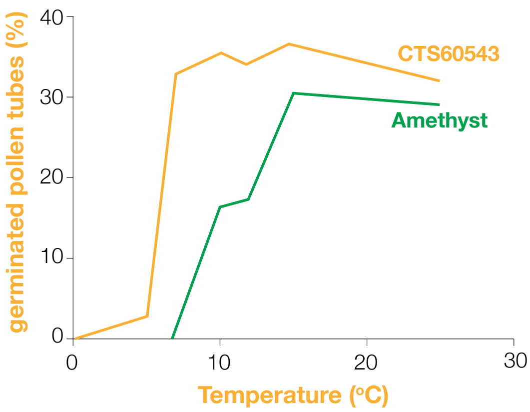

Figure 4: Proportion of pollen germination at various temperatures in cold-sensitive (Amethyst) and cold-tolerant (CTS60543) varieties.

At low temperatures pollen tubes grow slowly, fertilisation is less likely and the flower often aborts (Figure 4). The rate of pollen tube growth at low temperature is closely related to the cold tolerance of the whole plant (Figure 5). This trait can therefore be used to select more tolerant varieties.

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Experiments have also shown that the average of day and night temperatures is more important for flowering and podset, than any specific effects of either the maximum or the minimum temperature. 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 about 13°C are being developed.

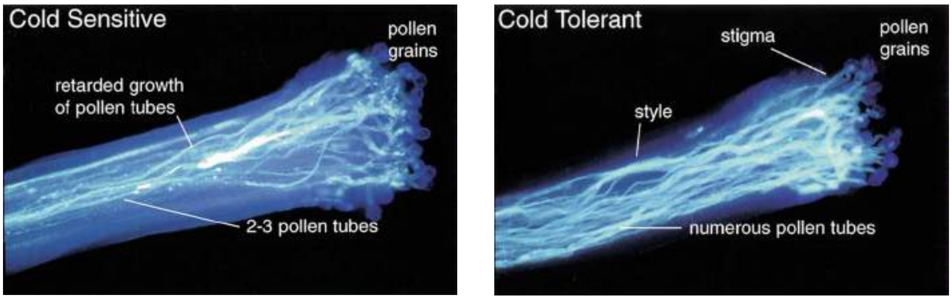


Figure 5: Pollen tube growth (stained with a fluorescent dye) in the stigma of cold-tolerant and cold-susceptible chickpea varieties. (Photo: H. Clarke, UWA)

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. [3]

More information

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2012/11/Using-RTK-steering-data-for-soil-erosion-control-and-water-logging-prevention

14.3 Waterlogging and flooding issues

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 (based on previous experience and paddock history). [4]

Observations from the wet season of 2010 indicate that the natural resistance all plants have to pathogens and pests is compromised when plants are stressed (from saturated conditions). In one trial in 2010 at Tamworth, Flipper⏀ had more Ascochyta blight than an adjoining plot of Yorker⏀. This is not what researchers expected. In another Tamworth trial, there was more Ascochyta blight in the wettest Kyabra⏀ plot than in better drained plots of Kyabra⏀, despite the fact that all had been sprayed eight times with 1.0 L/ha of chlorothalonil; therefore, stress from waterlogging reduced the ability to manage Ascochyta blight with a strategy that worked in plots that were less stressed. [5]

Symptoms of waterlogging can be confused with those of Phytophthora root rot but differ as follows (Table 1):

- Plants are most susceptible to waterlogging at flowering and early pod-fill.
- Symptoms develop within 2 days of flooding, compared to at least 7 days for Phytophthora root rot.
- 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.

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

[4] K Moore, M Ryley, M Sharman, J van Leur, L Jenkins, R Brill (2013) Developing a plan for chickpeas in 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

[5] K Moore, M Ryley, T Knights, P Nash, G Chiplin, G Cumming (2011) Chickpeas—varietal selection, paddock planning and disease management in 2011—Northern Region. GRDC Update Papers April 2011

More information

[Pulse Australia (2015). Chickpea: Managing Phytophthera root rot]

http://www.grdc.com.au/Research-and-Development/GRDC-Update-Papers/2013/02/Developing-a-plan-for-chickpeas-2013

http://link.springer.com/article/10.1007%2FBF02185569

http://link.springer.com/article/10.1007%2FBF02185570

Table 1: Differences between Phytophthora root rot 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 after a week or more	Symptom onset quite rapid
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

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. [6]

[6] K Moore, M Ryley, M Schwinghamer, G Cumming, L Jenkins (2011) Chickpea: Phytophthora root rot management. http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/phytophthora-root-rot

SECTION 15

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.

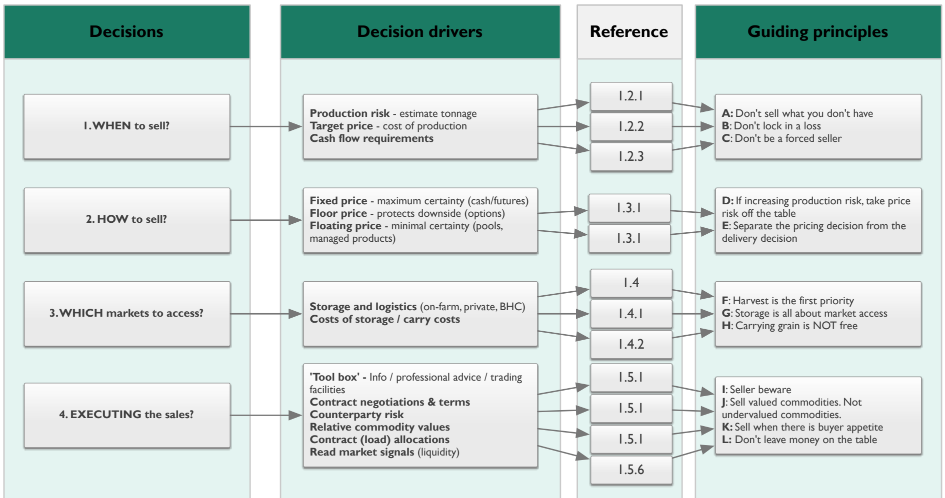
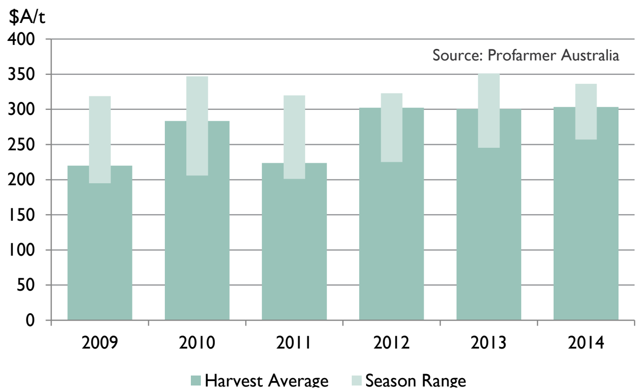


Figure 1: Grain selling flow chart. (Source. Profarmer Australia)

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

References are made to the section of the GrowNote you will find the detail.



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. (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 the target price and then work towards achieving that target price.

Unknowns include the amount of grain available to sell (production variability), the final cost of that production, 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 difficult to predict.

The skills growers have developed to manage production unknowns can be used to manage pricing unknowns. This guide will help growers manage and overcome price uncertainty.

15.1.1 Be prepared

Being prepared and having a selling plan is essential for managing uncertainty. The steps involved are forming a selling strategy and a plan for effective execution of sales.

A selling strategy consists of when and how to sell

When to sell

This requires an understanding of the farm's internal business factors including:

- production risk
- a target price based on cost of production and a desired profit margin
- business cash flow requirements

How to sell?

This is more dependent on external market factors including:

- Time of year determines the pricing method.
- Market access determines where to sell.
- Relative value determines what to sell.

The following diagram lists key selling principles when considering sales during the growing season.

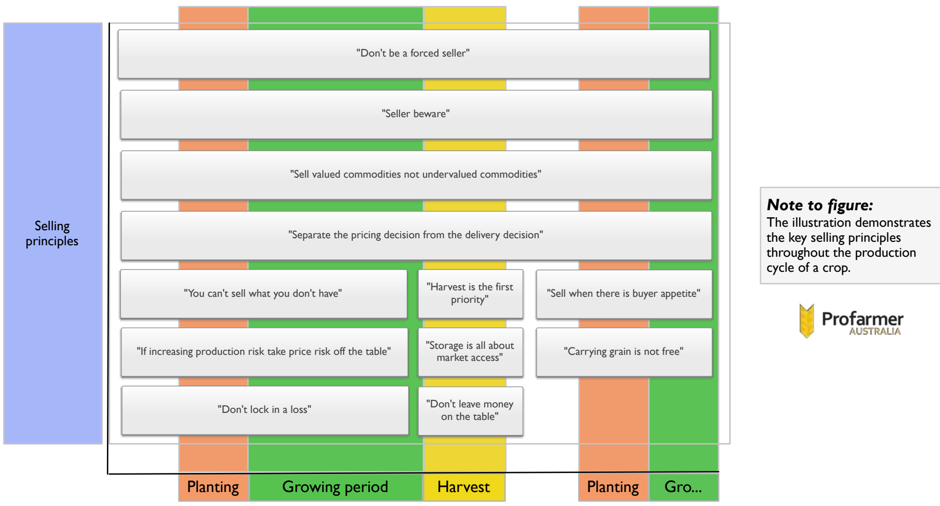


Figure 3: Grower commodity selling principles timeline. (Source. Profarmer Australia)

15.1.2 Establish the business risk profile (when to sell?)

Establishing your business risk profile allows the development of target price ranges for each commodity and provides confidence to sell when the opportunity arises. Typical business circumstances and how to quantify those risks during the production cycle are described below.

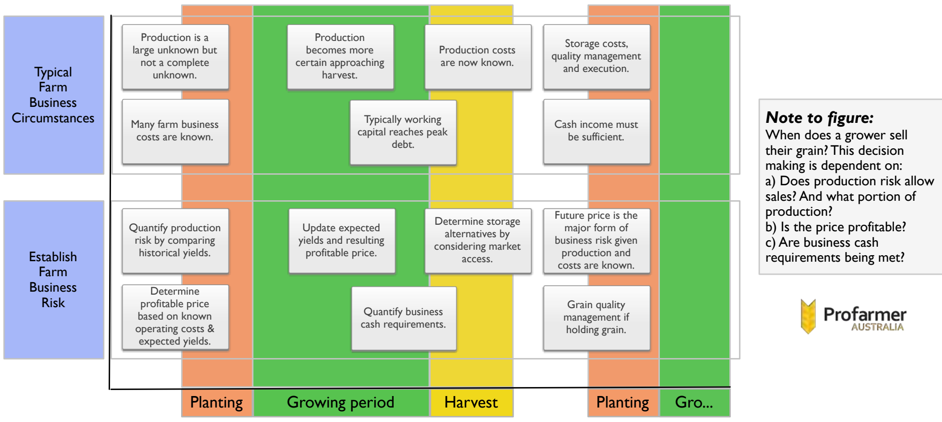


Figure 4: Typical farm business circumstances and risk. (Source. Profarmer Australia)

Production risk profile of the farm

Production risk is the level of certainty around producing a crop and is influenced by location (climate and soil type), crop type, crop management, and time of the year.

Principle: "You can't sell what you don't have" – Don't increase business risk by over committing production.

Establish a production risk profile by:

1. Collating historical average yields for each crop type and a below average and above average range.
2. Assess the likelihood of achieving average based on recent seasonal conditions and seasonal outlook.
3. Revising production outlooks as the season progresses.

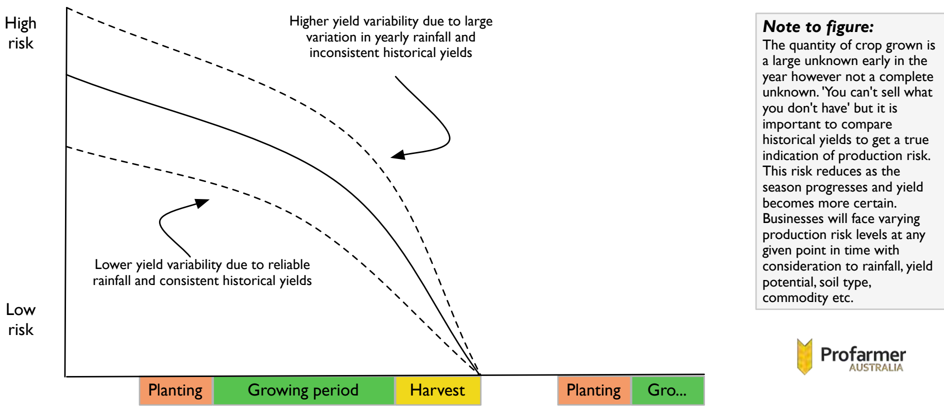


Figure 5: Typical risk profile of farm operation. (Source. Profarmer Australia)

Farm costs in their entirety, variable and fixed costs (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.

Principle: "Don't lock in a loss" – If committing production ahead of harvest, ensure the price is profitable.

Steps to calculate an estimated profitable price based on total cost of production and a range of yield scenarios is provided below.

Estimating cost of production - Wheat	
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
Freight to Port	$22 /t
Total per tonne costs	$48 /t
Cost of production Port track 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: GRDC's Farming the Business Manual also provides a cost of production templateand tips on grain selling vs grain marketing.

Income requirements

Understanding farm business cash-flow requirements and peak cash debt enables grain sales to be timed so that cash is available when required. This prevents having to sell grain below the target price to satisfy a need for cash.

Principle: "Don't be a forced seller" – Be ahead of cash requirements to avoid selling in unfavourable markets.

A typical cash-flow to grow a crop is illustrated below. Costs are incurred upfront and during the growing season with peak working capital debt incurred at or before harvest. This will vary depending on circumstance and enterprise mix. The second figure demonstrates how managing sales can change the farm's cash balance.

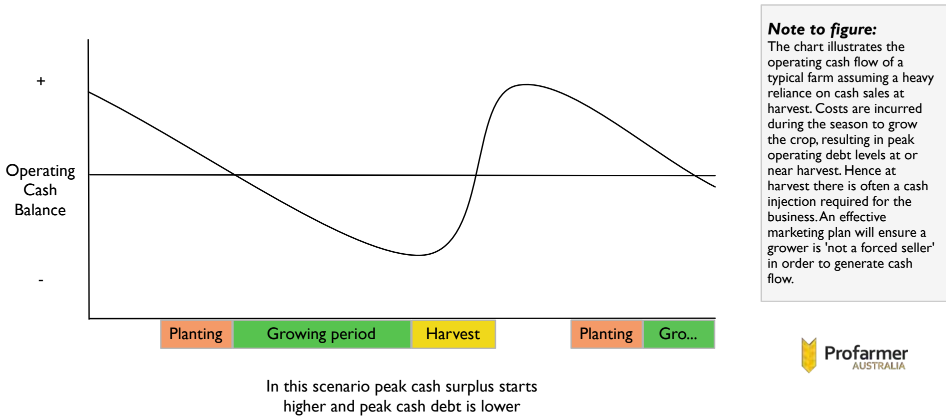


Figure 7: Typical operating cash balance (assuming harvest cash sales). (Source. Profarmer Australia)

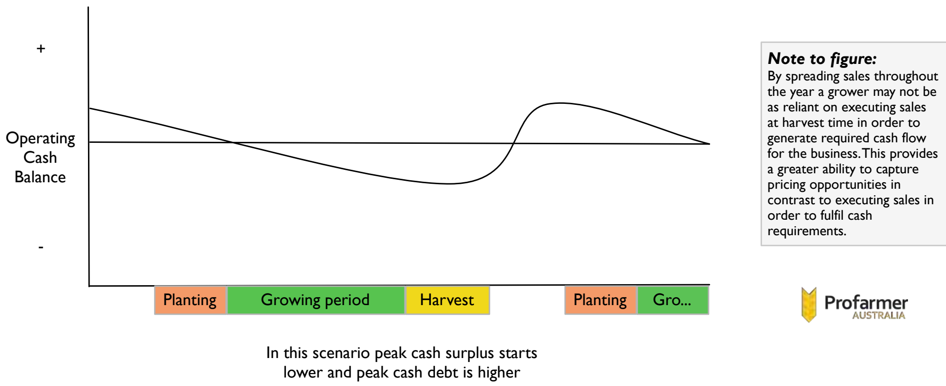


Figure 8: Typical operating cash balance cash sales spread throughout the year). (Source. Profarmer Australia)

When to sell revised

The “when to sell” steps above result in an estimated production tonnage and the risk associated with 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 (how to sell?)

The first part of the selling strategy answers the question “when to sell” and establishes comfort around selling a portion of the harvest.

The second part of the strategy addresses “how to sell”.

Methods of price management

Principle: “If increasing production risk, take price risk off the table” – When committing unknown production, price certainty should be achieved to avoid increasing overall business risk.

Table 1: Pricing products provide varying levels of price risk coverage.

	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

Principle: "Separate the pricing decision from the delivery decision" – Most commodities can be sold at any time with delivery timeframes negotiable, hence price management is not determined by delivery.

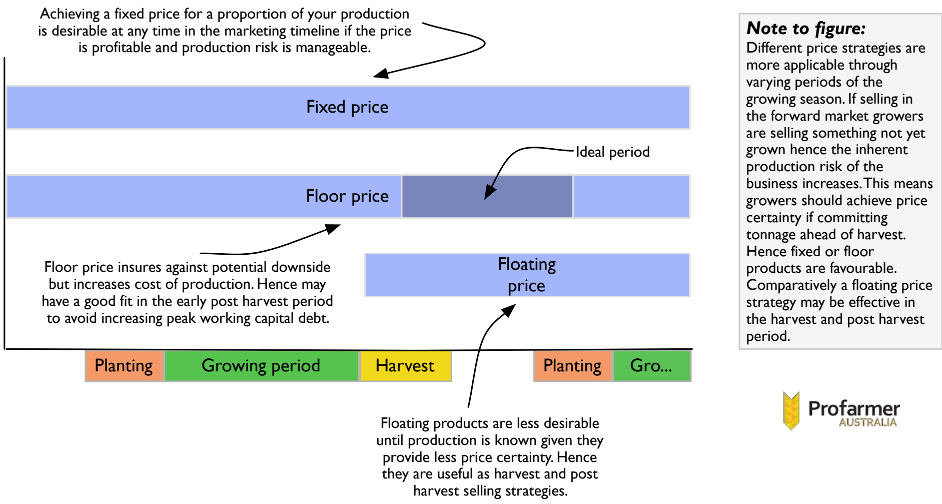


Figure 9: Summary of where different methods of price management are suited for the majority of farm businesses. (Source. Profarmer Australia)

Fixed price

A fixed price is achieved via cash sales and/or selling a futures position (swaps).

It provides some certainty around expected revenue from a sale as the price is largely a known except when there is a floating component in the price. For example, a multi-grade cash contract with floating spreads or a floating basis component on futures positions.

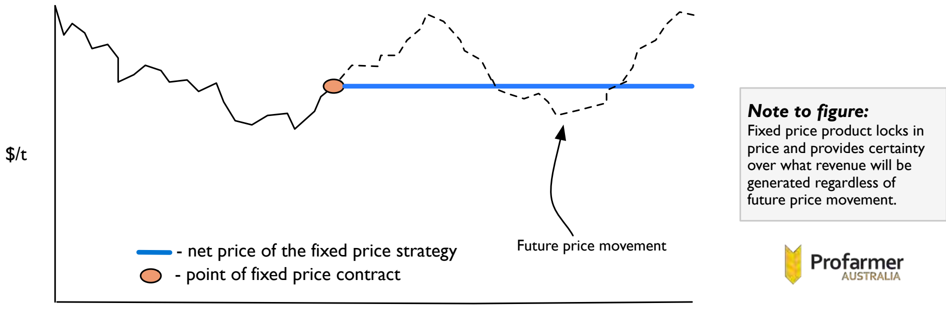


Figure 10: Fixed price strategy. (Source. Profarmer Australia)

Floor price

Floor price strategies can be achieved by utilising "options" on a relevant futures exchange (if one exists), or via a managed sales program product by a third party (ie. a pool with a defined floor price strategy). This pricing method protects against potential future downside whilst capturing any upside. The disadvantage is that the price 'insurance' has a cost which adds to the farm businesses cost of production.

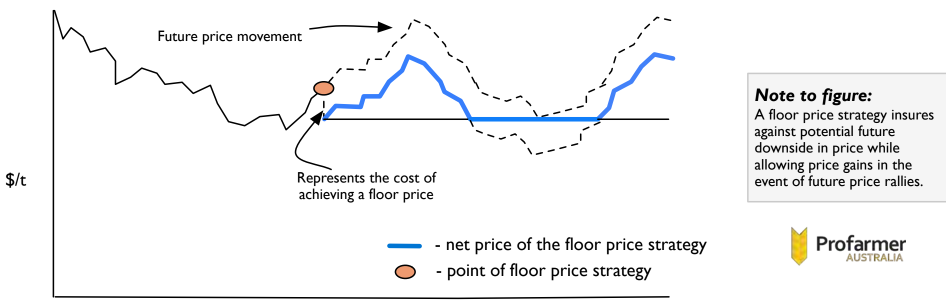


Figure 11: Floor price strategy. (Source. Profarmer Australia)

3. Floating price

Many of the pools or managed sales programs are a floating price where the net price received will move both up and down with the future movement in price. Floating price products provide the least price certainty and are best suited for use at or after harvest rather than pre harvest.

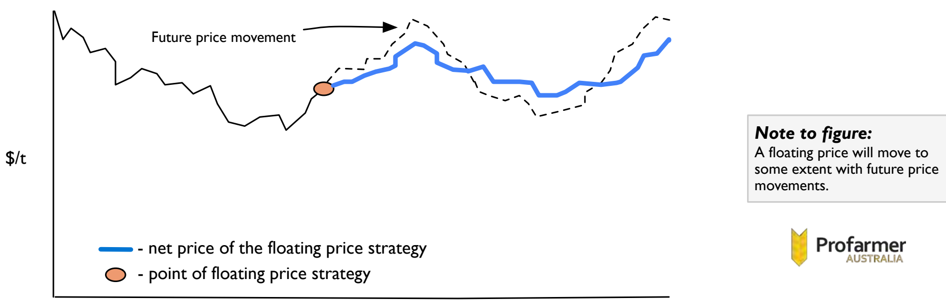


Figure 12: Floating price strategy. (Source. Profarmer Australia)

How to sell revised

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 are best used after harvest.

15.1.4 Ensuring access to markets

Once the selling strategy of when and how to sell is sorted, planning moves to storage and delivery of commodities to ensure timely access to markets and execution of sales. At some point growers need to deliver the commodity to market. Hence planning on where to store the commodity is important in ensuring access to the market that is likely to yield the highest return.

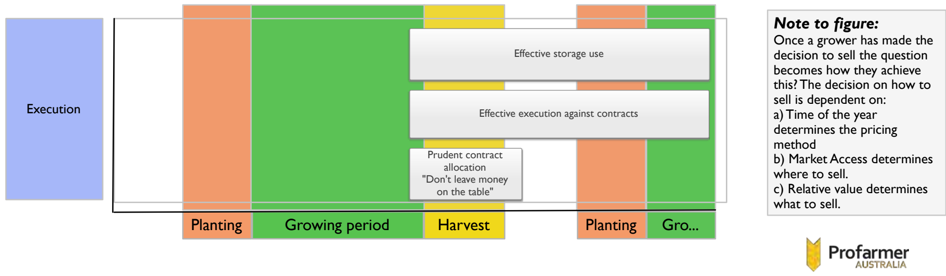


Figure 13: Effective storage decisions. (Source. Profarmer Australia)

Storage and Logistics

Return on investment from grain handling and storage expenses is optimised when storage is considered in light of market access to maximise returns as well as harvest logistics.

Storage alternatives include variations around the bulk handling system, private off farm storage, and on-farm storage. Delivery and quality management are key considerations in deciding where to store your commodity.

Principle: "Harvest is the first priority" – Getting the crop in the bin is most critical to business success during harvest, hence selling should be planned to allow focus on harvest.

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 feed lot, processor, or container packer), may be more suited to on-farm or private storage to increase delivery flexibility.

Storing commodities on-farm requires prudent quality management to ensure delivery at agreed specifications and can expose the business to high risk if this aspect is not well planned. Penalties for out-of-specification grain on arrival at a buyer's weighbridge can be expensive. The buyer has no obligation to accept delivery of an out-of-specification load. This means the grower may have to incur the cost of taking the load elsewhere whilst also potentially finding a new buyer. Hence there is potential for a distressed sale which can be costly.

On-farm storage also requires prudent delivery management to ensure commodities are received by the buyer on time with appropriate weighbridge and sampling tickets.

Principle: "Storage is all about market access" – Storage decisions depend on quality management and expected markets.

Reference:

For more information on on-farm storage alternatives and economics refer Section 14. Grain Storage.

For more information on on-farm storage alternatives and economics refer GRDC Western Region - Wheat - GrowNote, Chapter 14 Grain Storage

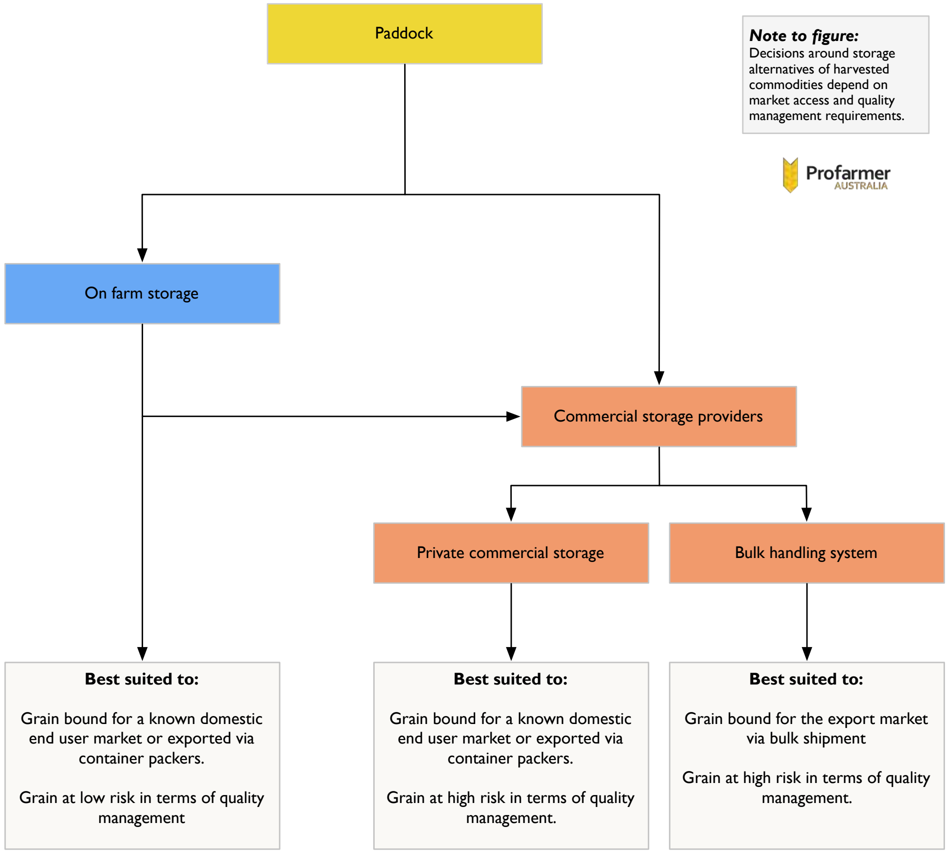


Figure 14: Grain storage decision-making. (Source. Profarmer Australia)

Cost of carrying grain

Storing grain to access sales opportunities post-harvest invokes a cost to "carry" grain. Price targets for carried grain need to account for the cost of carry.

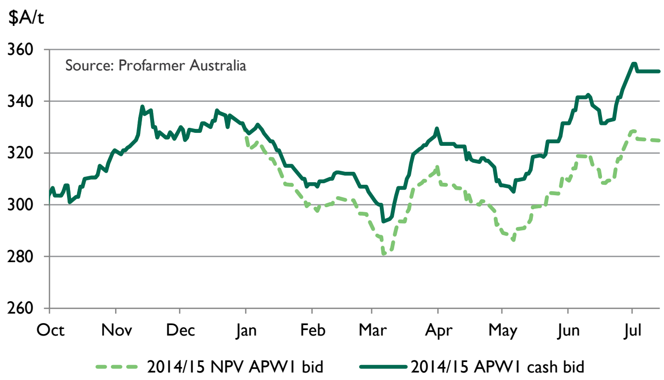
Carry costs are typically $3-4/t per month consisting of:

1. monthly storage fee charged by a commercial provider (typically ~$1.50-2.00/t per month)
2. the interest associated with having wealth tied up in grain rather than cash or against debt (~$1.50-$2.00/t per month 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 what was offered at harvest.

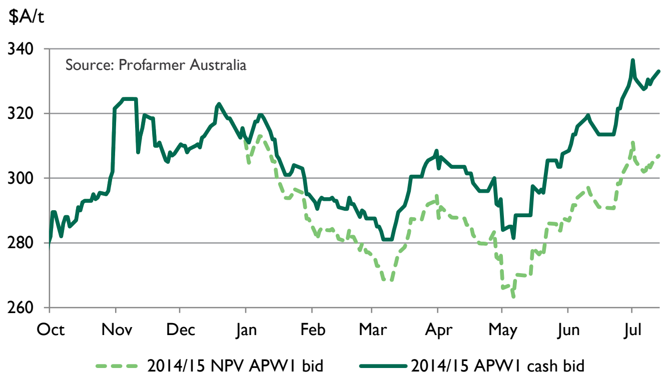
The cost of carry applies to storing grain on farm as there is a cost of capital invested in the farm storage plus the interest component. $3-4/t per month is a reasonable assumption for on farm storage.

Principle: "Carrying grain is not free" – The cost of carrying grain needs to be accounted for if holding grain and selling it after harvest is part of the selling strategy.



Note to figure:
If selling a cash contract with deferred delivery, a carry charge can be negotiated into the contract. For example in the case of a March sale of APW1 wheat for March-June delivery on buyers call at $300/t + $3/t carry per month, if delivered in June would generate $309/t delivered.

Figure 15: Brisbane APW2 cash vs NPV. (Source. Profarmer Australia)



Note to figure:
If selling a cash contract with deferred delivery, a carry charge can be negotiated into the contract. For example in the case of a March sale of APW1 wheat for March-June delivery on buyers call at $300/t + $3/t carry per month, if delivered in June would generate $309/t delivered.

Figure 16: Newcastle AWPI cash vs NPV. (Source. Profarmer Australia)

15.1.5 Ensuring market access revised

Optimising farm gate returns involves planning the appropriate storage strategy for each commodity to improve market access and cover carry costs in pricing decisions.

15.1.6 Executing tonnes into cash

This section provides guidelines for converting the selling and storage strategy into cash by effective execution of sales.

Set-up the tool box

Selling opportunities can be captured when they arise by assembling the necessary tools in advance. The toolbox includes:

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 service offerings and cost structures vary considerably. An effective grain selling professional will put their clients' best interest first by not having conflicts of interest and investing time in the relationship. Return on investment for the farm business through improved farm gate prices is obtained by accessing timely information, greater market knowledge and greater market access from the professional service.

1. Futures account and 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 CBOT can add significant value.

References:

The link below provides current financial members of Grain Trade Australia including buyers, independent information providers, brokers, agents, and banks providing over-the-counter grain derivative products (swaps).

http://www.graintrade.org.au/membership

The link below provides a list of commodity futures brokers.

http://www.asx.com.au/prices/find-a-futures-broker.htm

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:

- Price - Future price is largely unpredictable hence devising a selling plan to put current prices into the context of the farm business is critical to manage price risk.
- Quantity and Quality -When entering a cash contract you are committing to delivery of the nominated amount of grain at the quality specified. Hence production and quality risk must be managed.
- Delivery terms -Timing of 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 of grain to be transferred ahead of payment; hence counterparty risk must be managed.

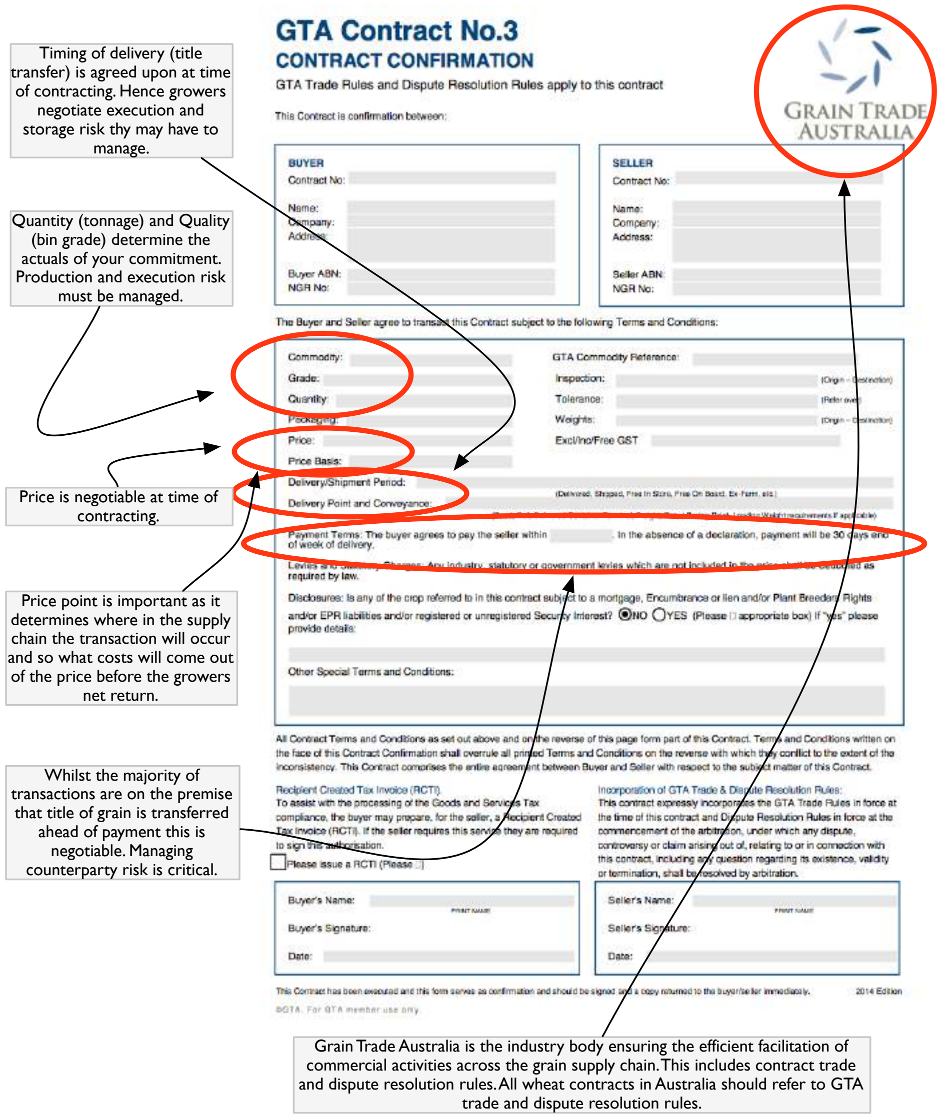


Figure 17: Typical cash contracting. (Source. Grain Trade Australia)

The price point within a cash contract will depend on where the transfer of grain title will occur along the supply chain. The below image depicts the terminology used to describe pricing points along the grain supply chain and the associated costs to come out of each price before growers receive their net farm gate return.

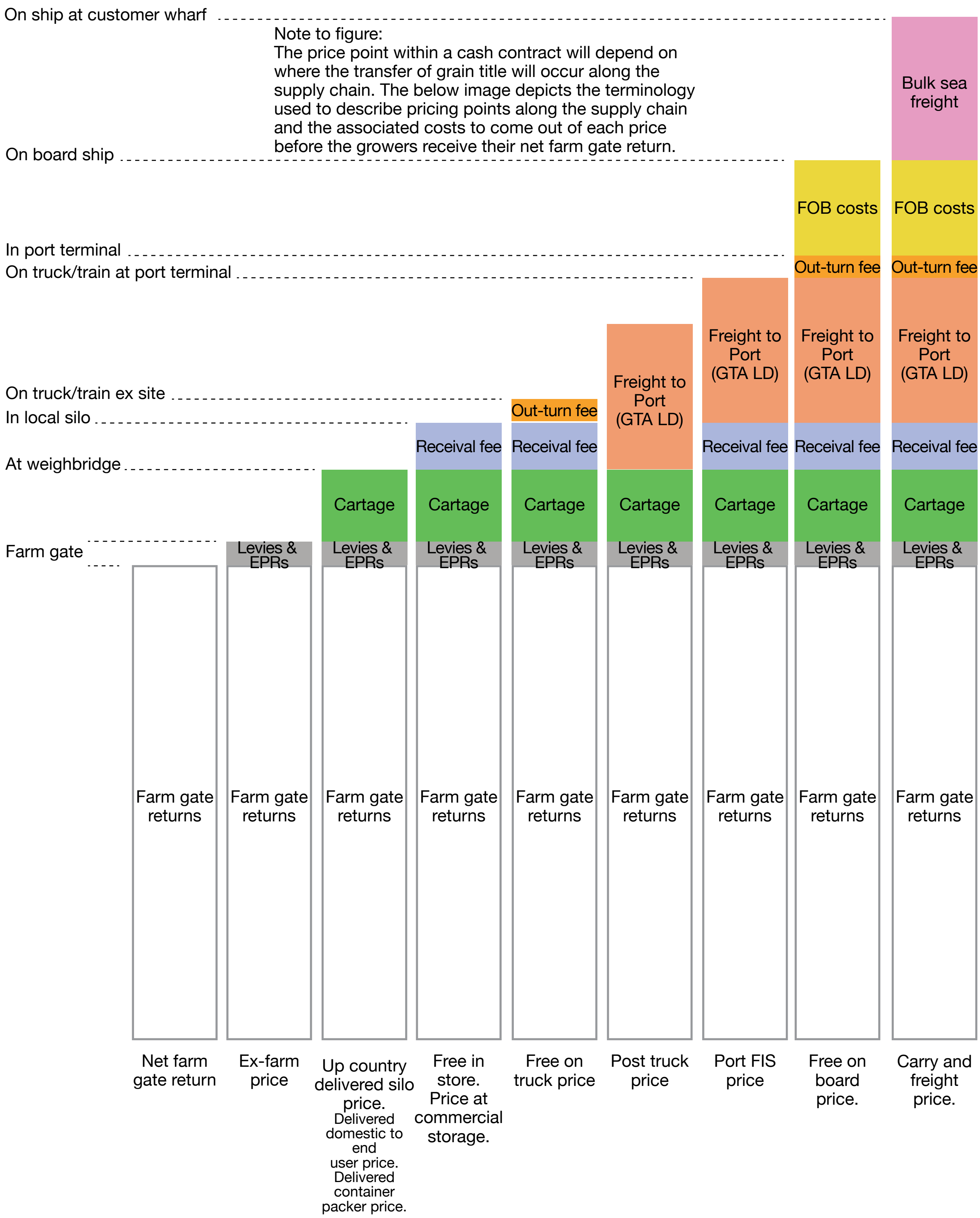


Figure 18: Cost and pricing points throughout the supply chains. (Source. Profarmer Australia)

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 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 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 firm offer and firm bid matches, the parcel transacts via a secure settlement facility where title of grain does not transfer from the grower until funds are received from the buyer. The availability of this 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.

References:

http://www.australiangrainexport.com.au/docs/Grain%20Contracts%20Guide.pdf

http://www.graintrade.org.au/contracts

http://www.graintrade.org.au/commodity_standards

http://www.graintransact.com.au

http://www.grainflow.com.au

http://emeraldgrain.com/grower-logins/

https://www.cleargrain.com.au/terms-and-conditions

https://www.cleargrain.com.au/get-started

Counterparty risk

Most sales involve transferring title of 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: "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.
- Conduct a credit check (banks will do this) before dealing with a buyer they are unsure of.
- Only sell a small amount of grain to unknown counterparties.
- Consider credit insurance or letter of credit from the buyer.
- Never deliver a second load of grain if payment has not been received for the first.
- Do not part with title of grain before payment or request a cash deposit of part of the value ahead of delivery. Payment terms are negotiable at time of contracting, alternatively the Clear Grain Exchange provides secure settlement where-by the grower maintains title of grain until payment is received by the buyer, and then title and payment is settled simultaneously.

Above all, act commercially to ensure the time invested in a selling strategy is not wasted by poor counterparty risk management. Achieving $5/t more and not getting paid is a disastrous outcome.

References:

GTA managing counterparty risk 14/7/2014 http://www.graintrade.org.au/sites/default/files/Grain%20Contracts%20-%20Counterparty%20Risk.pdf

Clear Grain Exchange title transfer model – https://www.cleargrain.com.au/get-started

GrainGrowers Guide to Managing Contract Risk www.graingrowers.com.au/policy/resources

Counterparty risk: A producer perspective, Leo Delahunty http://www.graintrade.org.au/sites/default/files/GTA_Presentations/Counterparty%20risk%20-%20a%20producer's%20perspective%20-%20Leo%20Delahunty.pdf

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 hold commodities that are not well priced at any given time. That is, give preference to the commodities of the highest relative value. This achieves price protection for the overall farm business revenue and enables more flexibility to a grower's selling program whilst achieving the business goals of reducing overall risk.

Principle: "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.

An example based on wheat and barley production system is provided below.

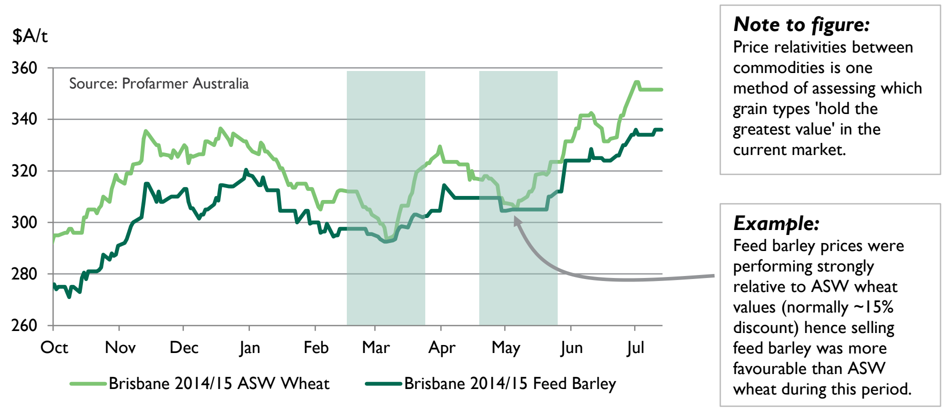


Figure 19: Brisbane ASW Wheat vs Feed Barley. (Source. Profarmer Australia)

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.

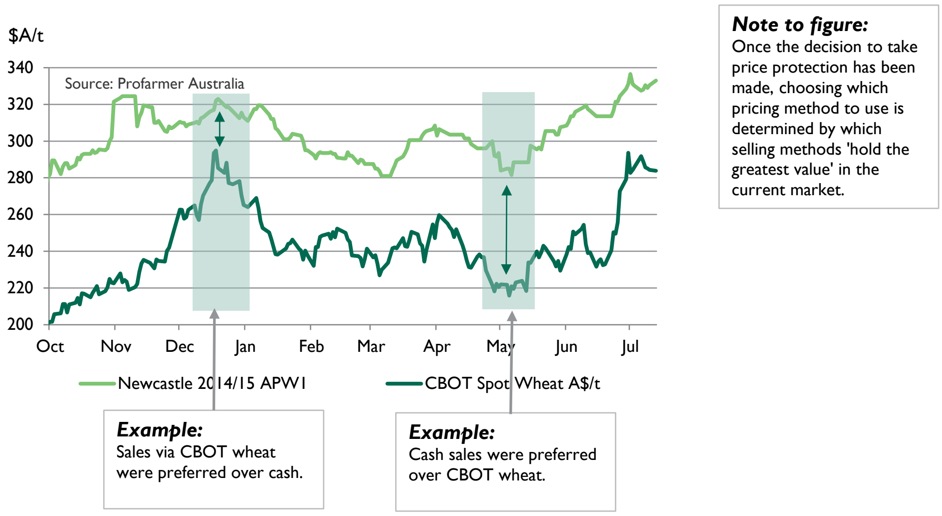


Figure 20: Newcastle ASWI vs CBOT Wheat A$/t. (Source. Profarmer Australia)

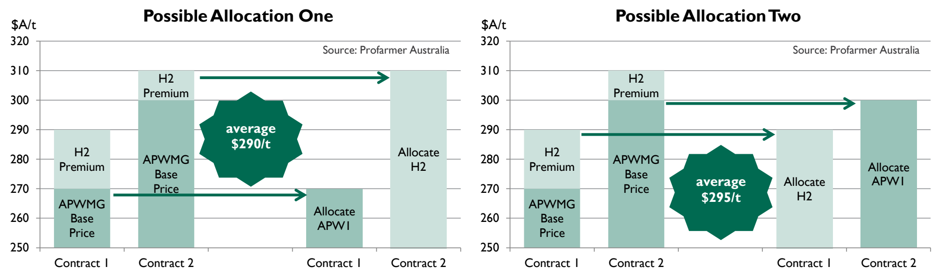
Contract allocation

Contract allocation means choosing which contracts to allocate your grain against come delivery time. Different contracts will have different characteristics (price, premiums-discounts, oil bonuses etc.), and optimising your allocation reflects immediately on your bottom line.

Principle: “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 wheat price growers should:

- Allocate your lower grades of wheat to contracts with the lowest discounts.
- Allocate higher grades of wheat to contracts with the highest premiums.



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 21: Possible allocation. (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 that interested in buying the commodity.

Principle: “Sell when there is buyer appetite” – When buyers are chasing grain, growers have more market power to demand a price when selling.

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 buyer appetite is strong. However if there is one buyer $5/t above the next best bid, it may mean cash prices are susceptible to falling $5/t if that buyer satisfies their buying 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.

15.1.7 Sales execution revised

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 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 prevent selling at a discount

15.2 Northern chickpeas – market dynamics and execution

15.2.1 Price determinants for northern chickpeas

Australia is a relatively small player in terms of world pulse production, producing 1-2 million tonnes of pulses in any given year vs 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 per cent.

There are two major types of chickpeas grown in Australia. The Desi chick pea is the predominant variety grown in NSW and Qld, while Kabuli is more prominent in South Australia and Victoria. The majority of the Desi chick pea crop is exported, and in terms of world trade Australia is a major player.

The major export markets for chickpeas are India and Pakistan, who between them import on average 1-1.5 million tonnes of chickpeas each year. In these markets field peas can also 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 find support 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 as Australia seeks alternate export destinations for local production.

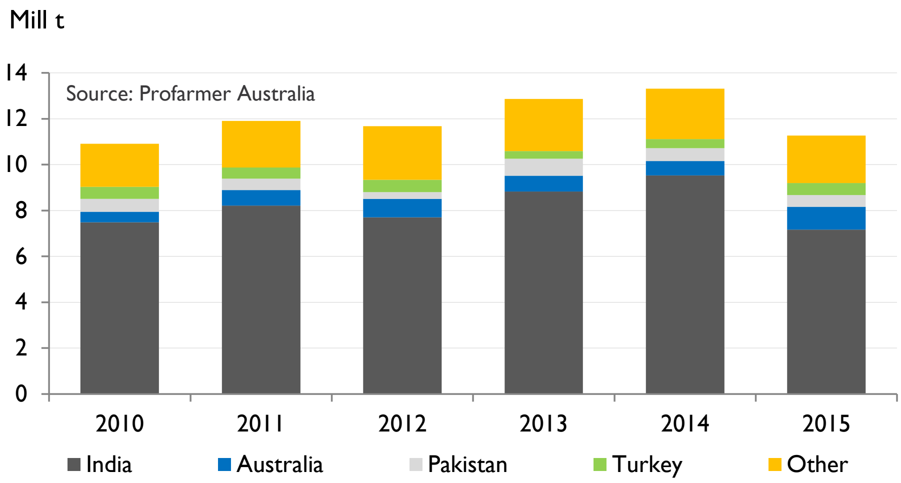


Figure 22: World chickpea production. (Source. Profarmer Australia)

Some of the global influences on Australian chick pea pricing are listed below:

1. Indian domestic Rabi season (Harvest April/May) pulse production. Any negative influences will increase the need for imports of either chickpea or field pea.
2. The world price of field pea. Field pea is purchased as a substitute pulse when the chickpea price is high.
3. Timing of festivals in importing countries. Ramadan is the most important festival which 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.

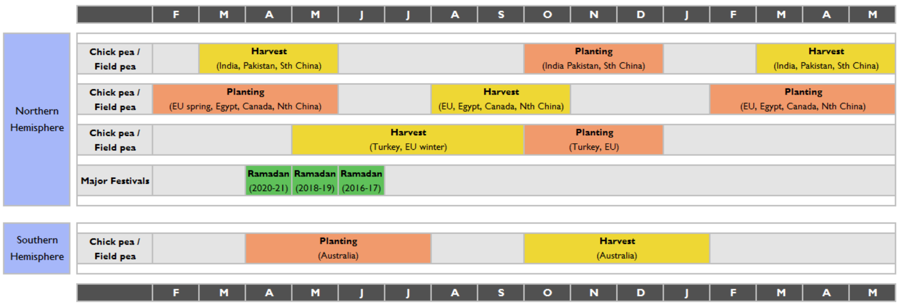


Figure 23: Global field pea and chickpea crop calendar.

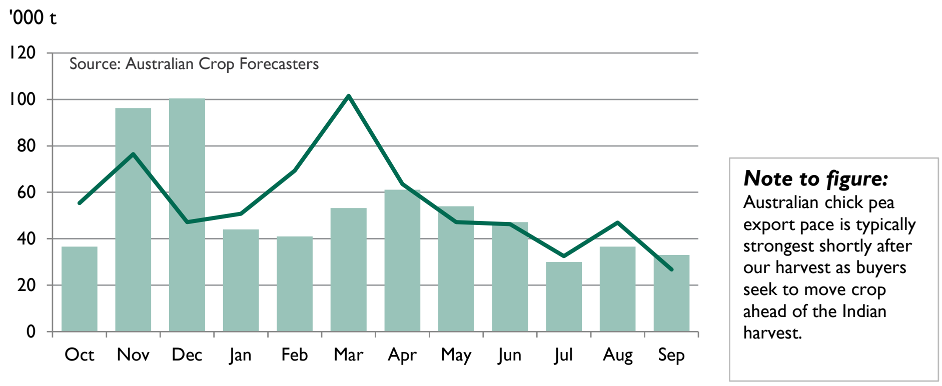


Figure 24: Monthly chickpea export pace. (Source. Australian Crop Forecasters)

15.2.2 Ensuring market access for Nothern chickpeas

The primary market for the northern desi chick pea crop is exports for human consumption. Of these exports approximately 30-40 per cent is exported in bulk and the remaining 60-70 per cent is exported in containers. The container or 'delivered' market can at times offer premiums to the bulk export market.

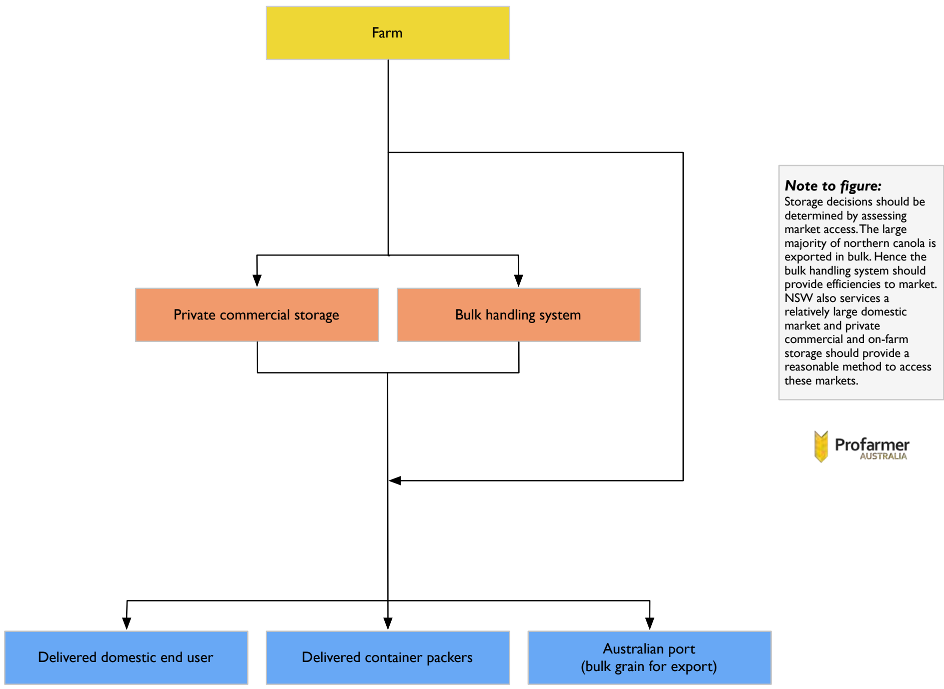
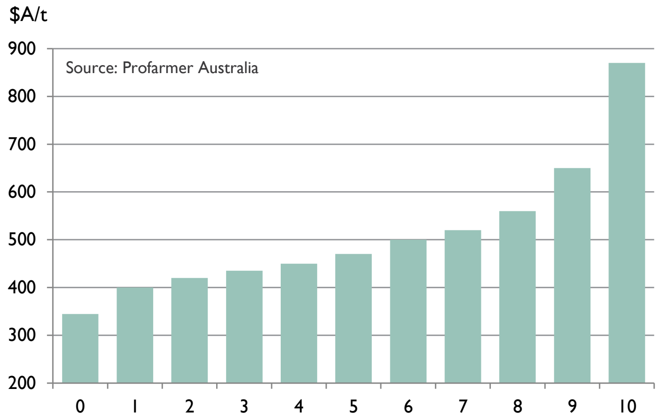


Figure 25: Australian supply chain flow. (Source. Profarmer Australia)

15.2.3 Executing tonnes into cash for Northern chickpeas

Given the volatile nature of chick pea pricing, setting a target price using the principles outlined in section 1.2.2 minimises the risk of taking a non-profitable price or holding out for an unrealistically high price that may not occur. Pricing deciles for chickpeas are provided as a guide.



Note to figure:
Decile charts such as the one to the left provide us an indication of how current values are performing relative to historical values. For example, a decile of 8 or above indicates current values are in the top 20% of historical price observations.

Figure 26: Brisbane chickpea decile. (Source. Profarmer Australia)

Selling options for chickpeas include:

1. Store on farm then sell: Most common occurrence. Chickpeas are relatively safe to store and require less maintenance than cereal grains. Must consider cost of storage in target pricing.
2. Warehouse then sell: this provides flexibility for sales if on farm storage is not available. Must consider warehousing costs in cost of production and target prices
3. Cash sale at harvest: least preferred option as buyer demand does not always coincide with harvest.

There are some forward price mechanisms available for chickpeas including area contracts as well as a traditional fixed volume forward contract. Whilst area based contracts tend to price at a discount to fixed volume contracts, this discount needs to be weighed up against the level of production risk inherent in each contract.

As with all sales, counterparty risk and understanding contract of sale is essential. Counterparty risk considerations is especially important for pulse marketing as there is often a higher risk of contract default in international pulse markets than for canola or cereals due to the markets they are traded into and lack of appropriate price risk tools (such as future). This can place extra risk on Australian based traders endeavouring to find homes for your product.

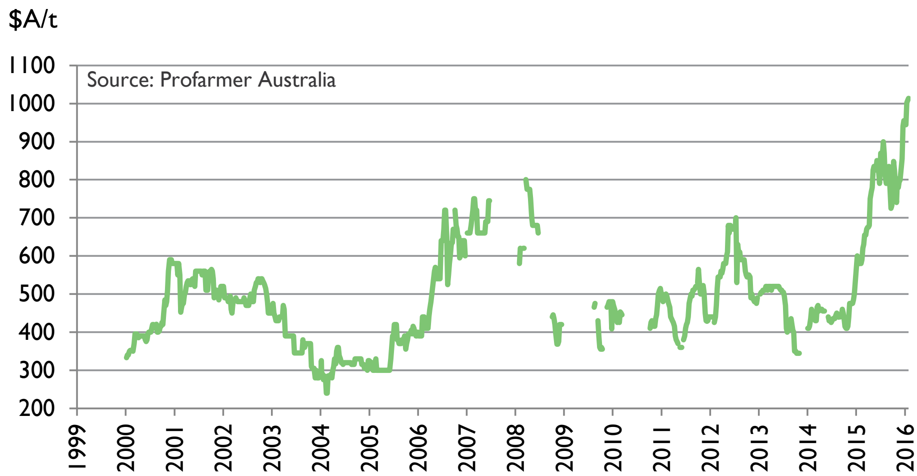


Figure 27: Brisbane chickpeas. (Source. Profarmer Australia)

SECTION 16

Current research

16.1 Searching GRDC projects

Each year the GRDC supports several hundred research and development, and capacity-building projects.

In the interests of improving awareness of these investments among growers, advisers and other stakeholders, the GRDC has assembled summaries of projects on its website.

These summaries are written by GRDC's research partners as part of the Project Specification for each project, and are intended to communicate a useful summary of the research activities for each project investment.

The review expands existing communication products where we summarise the R&D portfolio in publications such as the Five-year Strategic Research and Development Plan, the Annual Operating Plan, the Annual Report and the Growers Report.

GRDC's project portfolio is dynamic with projects concluding and new projects commencing on a regular basis.

The GRDC values the input and feedback it receives from its stakeholders and so would welcome your feedback on this website search engine.

Visit http://projects.grdc.com.au/search.php

SECTION 17

Key contacts

GRDC Northern Regional Panelists

John Minogue - Chair

John Minogue runs a mixed broadacre farming business and an agricultural consultancy, Agriculture and General Consulting, at Barmedman in south-west NSW. John is chair of the local branch of the NSW Farmers' Association, has formerly sat on the grains committee of the NSW Farmers' Association and is a winner of the Central West Conservation Farmer of the Year award. John has also been involved in the biodiversity area as a board member of the Lachlan Catchment Management Authority. His vast agricultural experience in central west NSW has given him a valuable insight into the long-term grains industry challenges.
M 0428 763 023 E jlminogue@bigpond.com

Penny Heuston - Deputy Chair

Penny Heuston is an agronomist based in Warren, NSW. She is passionate about the survival of the family farm and its role in the health of local economies. Penny is dedicated to ensuring research is practical, farm-ready and based on sound science and rigor. She sees 'two-way communication' as one of the panellists' primary roles and is committed to bringing issues from the paddock to 'the lab' and conversely, the science to the paddock.
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Loretta Serafin

Loretta Serafin has extensive experience as an agronomist in northwest NSW and works with the NSW Department of Primary Industries in Tamworth. As the leader northern dryland cropping systems, she provides expertise and support to growers, industry and agronomists in the production of summer crops. Loretta is a member of numerous industry bodies and has a passion for helping growers improve farm efficiency. She sees her role as a conduit between advisers, growers and the GRDC to ensure growers' research needs are being met.
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- Australian Pulse Standards 2014/2015, Pulse Australia. https://www.graintrade.org.au/sites/default/files/file/Commodity%20Standards/Pulse%20Standards%20201415%20Final.pdf

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