GRDC Grains Research Update

Grains Research UPDATE

NARRABRI

Friday 22nd July 2016

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GRDC Grains Research Update Narrabri

Friday 22nd July, 2016, The Crossing Theatre

8:30am registration for a 9:00am start, finish 3:00pm

Agenda

Time	Topic	Speaker (s)
9:00 AM	Welcome	GRDC
9:10 AM	**Crown-rot resistance breeding**	*Phil Davies (University of Sydney) & Meiqin Lu (AGT)*
9:45 AM	**Chickpea and faba bean disease update**.	*Kevin Moore (NSW DPI)*
10:10 AM	**A new strain of wheat leaf rust.** Potential impacts, which varieties and what to look for. **Adult plant resistance** - its role and use in rust management.	*Robert Park (University of Sydney PBI, Cobbitty)*
10:40 AM	Morning tea	
11:10 AM	**Killing storage pests without mercy** – fumigation strategies that work and **Super cool results -** achieving great results with silo aeration.	*Pat Collins & Philip Burrill (DAF Qld)*
11:50 AM	**Russian Wheat Aphid** – identification, detection, management and implications	*Melina Miles (DAF Qld)*
12:15 PM	**Managing patches of glyphosate resistant weeds** – what have we learnt from grower experience? Costs, advantages and practicalities of key IWM tactics.	*Tony Cook (NSW DPI)*
12:40 PM	Lunch	
1:40 PM	**Tillage impacts of weed seed burial and subsequent management**.	*Michael Widderick (DAF Qld)*
2:05 PM	**Harvest weed seed capture systems in the northern region.** Experience with the Harrington Seed Destructor and the Chaff Deck system for weed tramlining.	*Michael Walsh (University of Sydney)*
2:30 PM	**Stopping spray drift - volatility, droplet drift, inversions and management.** Field and wind tunnel studies, comparing spray deposition and drift potential of a range of nozzles and droplet size spectra all tied together with automated weather updates by a new App.	*Chris O'Donnell (University of Queensland)*
3:00 PM	Close	

Contents

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Resistance and tolerance: where we are with crown rot breeding

Philip Davies, University of Sydney

Key words

Crown rot, pre-breeding, varietal selection

GRDC code

US00075

Take home message

- Significant variation exists in varieties performance under crown rot, representing opportunities for intervention in crown rot management.
- Varietal performance under crown rot is made up separately of resistance and tolerance, as well as the yield potential of the variety.
- The current simplistic R to S resistance rating system does not adequately represent a variety's true performance in crown rot conditions, as it neglects both the tolerance and yield potential of the variety.
- A more informative rating system for crown rot is required which accounts for a variety's resistance and tolerance to crown rot, and the variety's yield potential.

Background

Crown rot caused by the fungus *Fusarium pseudograminearum* is a major limiting factor in winter cereal production in the northern region, and continues to emerge as a serious issue in the southern and western regions. The disease is characterised by a light to dark honey-brown discolouration on the base of infected tillers, extending further up the stem in more susceptible varieties. This discolouration is directly related to the invasion of the plant tissue by the pathogen during the infection phase of this disease.

As the pathogen continues to colonise the plant, fungal growth begins to disrupt vascular tissues, interrupting water and nutrient flow up and down tillers. With the increased water and nutrient demands placed on the plant during the post-flowering and grain-fill period, this disruption results in premature ripening of affected heads, causing the typical whiteheads and high screenings. Moisture and heat stress around this period greatly increase the expression of the disease through increased water demands placed on the plant.

The pathogen survives as mycelium (fungal growth) in winter cereal and grass weed residues which had become infected during the season. Whilst stubble breakdown (and thus pathogen survival) is strongly associated with soil temperature and moisture conditions, as long as there is intact cereal residues, viable crown rot inoculum will be present.

Yield losses associated with crown rot can be significant, and while there are no silver bullet solutions to this disease, management strategies and genetic variation within existing varieties are available.

Management options for crown rot generally are associated with reducing the incidence of infection. This includes rotation to non-host crops, often used in conjunction with risk management strategies such as PreDictaB, inter-row sowing, and grass weed management. Whilst these strategies are

effective at reducing the incidence of infection, once a plant becomes infected, the resistance and tolerance of the variety determines how the plant responds.

Describing the impact of crown rot

To look at the impact of crown rot, Figure 1 summarises the yield loss experienced in a subset of varieties from a breeding trial in 2015. This suite of varieties represents the range of responses to crown rot, from highly susceptible to resistant, and from intolerant to tolerant.

This trial achieved moderate disease pressure, with approximately 80% of plants infected. The susceptible variety EGA Gregory[Ⓓ] lost 28% of its yield due to crown rot, compared to Sunguard[Ⓓ], which lost 12%. From these figures, it is clear that Sunguard[Ⓓ] performs better under crown rot than Gregory[Ⓓ]. This however does not describe why this difference exists.

A varieties performance under crown rot is determined by both resistance and tolerance. These are two separate traits which together determine yield loss. Resistance is the ability of a plant to restrict the infection by the pathogen, or restrict its growth throughout the plant. Tolerance on the other hand is the ability of the plant to yield, despite being infected. Resistance is widely discussed with regards to crown rot, and used to describe how a variety will perform under crown rot (variety sowing guides etc.). Importantly, tolerance equally impacts a varieties response to crown rot but is often not considered.

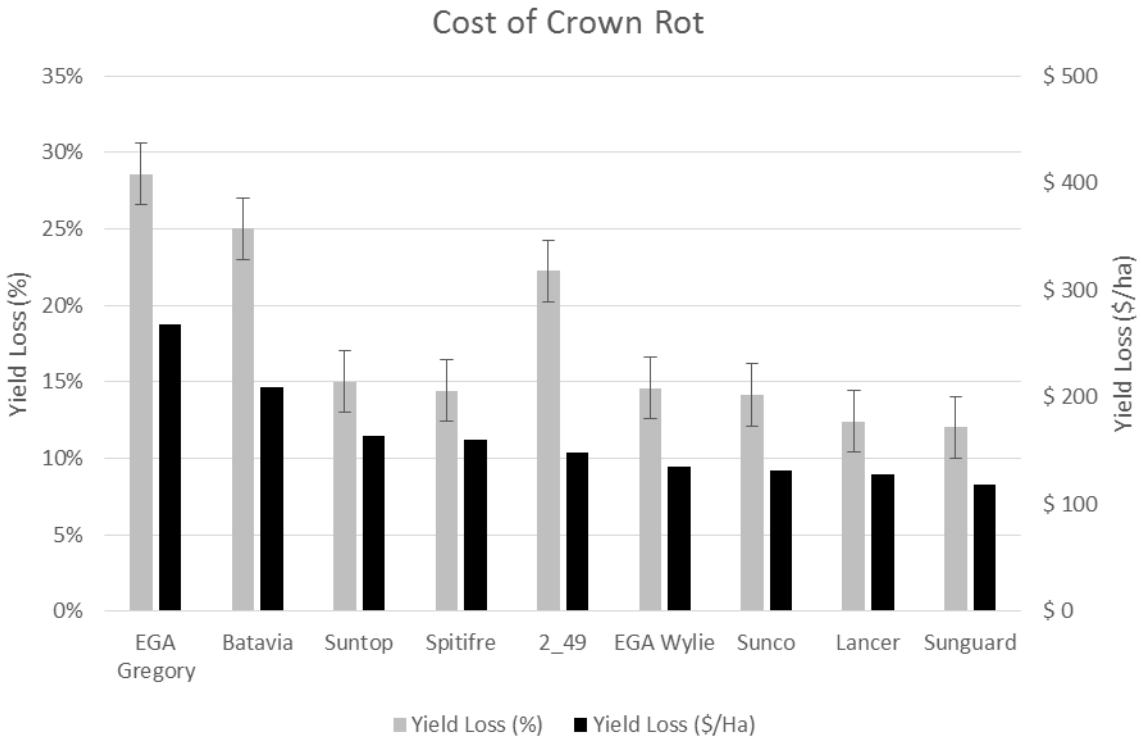

Figure 1. Yield loss of varieties associated with crown rot, described as both a percentage and in dollar terms, calculated at $300/tonne.
Ⓓ Varieties EGA Gregory, Suntop, Spitfire, EGA Wylie, Lancer and Sunguard are protected under the Plant Breeders Rights Act 1994.

Whilst reporting both resistance and tolerance provides more information from which to base varietal selections, it does not tell the whole story, as it fails to take into account the inherent yield potential of these varieties.

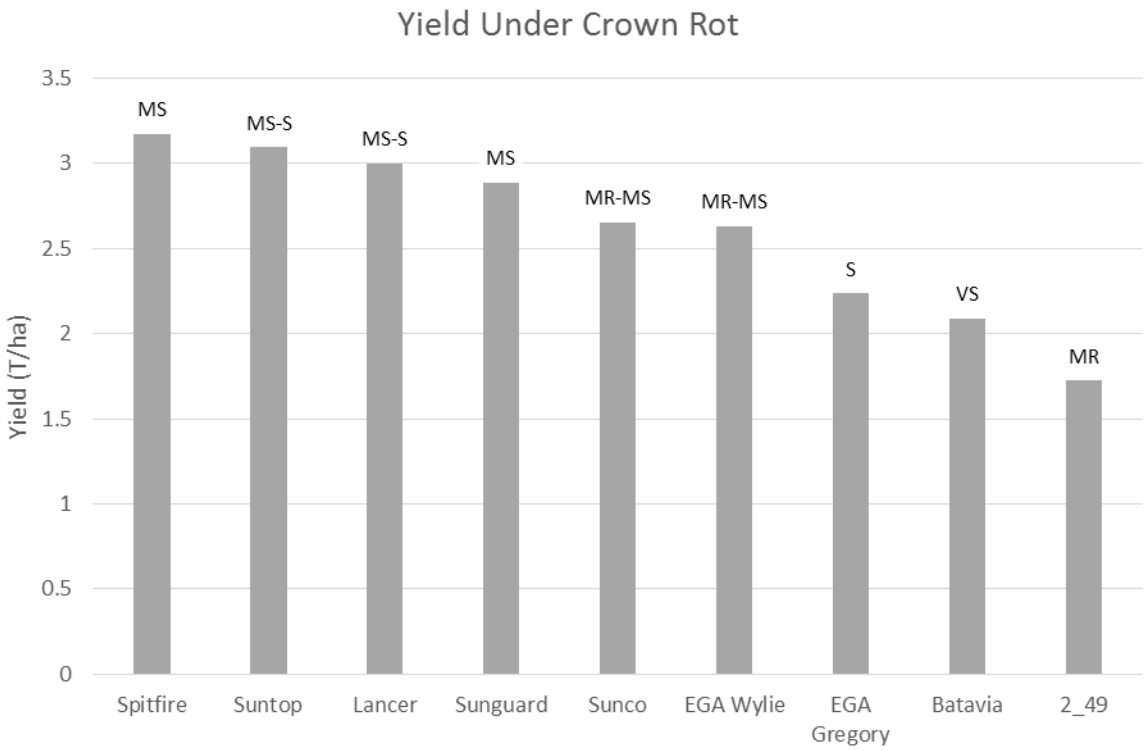

Figure 2. Yield of a suite of wheat varieties under moderate levels of crown rot.
Varieties Spitfire, Suntop, Lancer, Sunguard, EGA Wylie and EGA Gregory protected under the Plant Breeders Rights Act 1994.

This is demonstrated in Figure 2, comparing the yield under crown rot pressure in the same suite of varieties. In this case, even though Sunguard has a lower yield loss, Spitfire, Suntop and Lancer all out performed it under moderate crown rot conditions. The financial impact becomes even more pronounced when you consider that Sunguard is an AH variety compared to the APH classifications of Spitfire, Suntop and Lancer.

While the resistance rating system may be more effective under very high inoculum conditions, under the moderate disease pressure achieved in this trial, the most resistant varieties of EGA Wylie and Sunco were significantly outperformed, and these ratings risk misinforming growers of the true performance of varieties under crown rot conditions.

Varietal selection with respect to crown rot should therefore consider not only the resistance rating of a variety, but also its tolerance and yield potential, along with the level of crown rot inoculum and level of risk a grower is willing to take. Soil starting moisture as a buffer against the potential effects of water and heat stress on disease expression should also factor in decision making.

How do pre-breeders use this information?

Resistance and tolerance represent two distinct genetic traits which can be targeted to reduce the impact of crown rot. By separately assessing both these traits, they can be combined to achieve an additive effect.

The level of resistance of a variety can be assessed by examining the degree of honey-brown discolouration on the lower stem and crown. While time-consuming, this strategy is useful in determining the amount of fungus in the plant, and thus the relative level of susceptibility. Tolerance however is measured in yield loss trials, comparing the amount of yield loss of a breeding line to the amount of stem browning in that line. This allows breeders to identify whether a low yield loss amount can be attributed to resistance or tolerance in that line.

While these trials are useful in a smaller pre-breeding context where accurate identification of crown rot traits is important, they are both too time consuming and expensive to use in a commercial breeding program.

Breeding for crown rot in a commercial context

The use of molecular markers can alleviate some of these issues, by providing breeders with a rapid method of determining whether certain genes or combination of genes are present in a breeding line which confer improved resistance or tolerance to crown rot. Whilst molecular markers are particularly effective for some traits, markers associated with crown rot have some difficulties. There are only a few markers available for a small number of resistance QTLs only, and these are often unreliable, and only loosely linked with the trait.

These issues are being addressed, with recent work identifying a number of markers associated with tolerance traits in a mapping population. These markers will make it easier to breed for tolerance traits without having to resort to yield loss testing. Further, refinement of existing markers for some of the key resistance QTLs is being completed by CSIRO in Brisbane, which will result in more reliable and thus effective in commercial breeding programs.

Along with work on molecular markers, there are also efforts to improve the screening methodologies for resistance and tolerance. Current screening methods are very time consuming and thus expensive to run and have relatively low throughput. Research currently being undertaken at The University of Southern Queensland looking at remote sensing technologies is aiming to improve the screening process. This includes using multispectral imagery to detect differences between resistant and susceptible genotypes, as well as using infrared imagery to look at differences in canopy temperature between tolerant and intolerant lines, as a way of determining plant moisture stress.

Conclusion

Accurate measurement and classification of a varieties performance under crown rot is crucial for varietal selection. This should include resistance, tolerance and the yield potential of a variety. This is similarly the case for breeders, both in a pre-breeding and commercial context, where the resistance and tolerance separately need to be measured, so that these traits can be combined.

Acknowledgements

This research is a collaborative project between The Universities of Sydney and Southern Queensland, and the CSIRO. The research undertaken as part of this project is made possible by the significant contributions of growers through both trial cooperation and the support of the GRDC, the author would like to thank them for their continued support.

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Chickpeas – what we learnt in 2015 and recommendations for 2016

Note: Recommendations for Ascochyta were revised in May 2016 – please see related article in these proceedings

Kevin Moore, Leigh Jenkins, Paul Nash, Gail Chiplin and Sean Bithell, Department of Primary Industries, NSW

Key words

Chickpea, Ascochyta, Phytophthora, management

GRDC code

DAN00176 Northern NSW Integrated Disease Management

Take home message

- Plant seed of known identity and purity and of high quality that has been properly treated with a registered seed dressing.
- Localities where Ascochyta was found on any variety in 2015 are considered high risk for 2016 crops and growers are advised to apply a preventative fungicide before the first post-emergent rain event to all varieties including PBA HatTrick®.
- Mild temperatures, long cloudy periods and frequent rainfall events during Jun/Jul across the Northern region as occurred in 2015, are ideal for early season outbreaks of Ascochyta blight in chickpea crops.
- In wet seasons the management of Ascochyta can be hindered by getting ground rigs into wet paddocks and shortage of fungicides.
- Follow the disease management recommendations in this article and associated links – they will maximise your chance of a profitable chickpea crop in 2016.

The 2015 northern NSW/southern QLD chickpea season

Unprecedented high prices (peaking at $900 in Jun) led to a record planting of chickpeas in the region. The 2015 winter crop season in northern NSW/southern QLD followed a wet Jan, dry Feb/Mar, wet Apr (except Dalby) and wet May (except Roma, Table 1).

In most centres in northern NSW, mild, wet to very wet conditions in Jun/Jul were followed by average or below average Aug, a very dry Sep, below average Oct rain and a wet Nov harvest. On the Downs conditions were much drier. Rainfall totals and long term averages for the Jun-Nov period were: Dubbo 292mm (LTA 279mm), Gilgandra 301mm (LTA 261mm), Trangie 251mm (LTA 225mm), Nyngan 204mm (LTA 190mm), Coonamble 158mm (LTA 231mm), Walgett 236mm (LTA 201mm), Moree 204mm (LTA 258mm), Tamworth 341mm (LTA 315mm), Roma 173 (LTA 226mm), Dalby 124mm (LTA 261mm) with monthly figures in Table 2.

With the exception of the Downs and western areas, these conditions, together with early sowing resulted in high biomass crops which used a lot of water. Cold, dry weather from late August to late September led to flower and pod abortion. This was not helped by considerable temperature fluctuations in the last 10-14 days of September (up to 20°C in a 24hr period). Hot, dry conditions in early October put crops under further stress (as most had run out of water). Thus, in many parts of northern NSW, seasonal conditions conspired to produce big canopies that ran out of water during the major pod filling period. Coupled with frosts, low and fluctuating temperatures, this resulted in missing pods, ghost pods or single-seed pods.

Table 1. Jan – May 2015 rain (mm) at selected locations in NSW/QLD

Location	Jan	Feb	Mar	Apr	May
Roma	86	31	33	46	12
Dalby	107	49	13	11	86
Dubbo	131	32	8	82	48
Gilgandra	103	21	3	99	73
Trangie	59	1	11	114	48
Nyngan	91	5	13	44	44
Coonamble	74	11	6	76	51
Walgett	34	0	6	24	30
Moree	105	4	60	63	33
Tamworth	90	23	52	86	38

Table 2. Jun – Nov 2015 rain (mm) at selected locations in NSW/QLD

Location	Jun	Jul	Aug	Sep	Oct	Nov
Roma	64	12	24	16	16	41
Dalby	10	18	24	15	47	9
Dubbo	72	60	39	8	46	67
Gilgandra	87	59	31	1	32	92
Trangie	44	44	33	3	28	99
Nyngan	51	35	29	7	13	70
Coonamble	39	27	13	4	29	35
Walgett	58	44	27	1	34	72
Moree	62	36	11	4	10	83
Tamworth	109	34	54	24	50	71

Nevertheless, in NSW yields east of the Castlereagh and Newell highways were generally good with the better crops going 2.5 – 3.0 t/ha. However, farmers west of these highways were disappointed with some crops yielding less than 0.2 t/ha.

In QLD, some crops on the Downs planted on wide rows went >3.0 t/ha with at least one Kyabra® crop going 3.6 t/ha. The Downs crops were sown on a full profile but with in-crop rainfall well below average, they did not have a lot of biomass. This, coupled with wide rows which allowed the soil to warm up, is believed to account for the large yield differences between crops at say Dalby and those at Moree.

Chickpea diseases in 2015

In 2015, 243 crop inspections were conducted as part of DAN00176. Ascochyta blight, AB (*Phoma rabiei* formerly called *Ascochyta rabiei*) was detected in 60 crops. High chickpea prices tempted some growers to break rules, eg plant back to back chickpeas and they paid the price, in terms of AB infection and AB management costs in 2015 chickpea crops that followed 2014 chickpeas. Some growers reported more AB in PBA HatTrick® than they ever saw in Jimbour, but many of these crops had been inundated in Jun/Jul and we know that AB resistance of waterlogged chickpeas is compromised. Further the genetic purity of the variety could not be determined. Generally, however, good management and dry conditions through Aug – Oct kept AB under control and no major yield losses were reported.

Phytophthora root rot, PRR (*Phytophthora medicaginis,* 23 cases) caused light to moderate losses but only in paddocks with a history of medics or where the susceptible variety PBA Boundary® was planted.

The mild wet winter also favoured Sclerotinia (24 cases) especially in paddocks with a canola history, with both basal and aerial infections detected. Where canola was involved, the species was always *S. sclerotiorum*. One crop in the wetter areas east of Narrabri had aerial infection from ascospores of *S. minor* instead of the typical infection of roots and stem base by mycelia from sclerotia. This was the first record in this region for infection from windborne ascospores from sclerotia (due to carpogenic germination of sclerotia) leading to infection of chickpea by of *S. minor*. If such windborne infection is common, greater *S. minor* infection may result.

Botrytis Grey Mould, BGM (*Botrytis cinerea*) threatened to be a problem in high biomass crops and some of these were sprayed with carbendazim in early spring. This together with the hot dry finish, diminished the risk of BGM and no damage was reported.

Across the region, viruses were uncommon only reaching damaging levels in crops with poor, patchy stands (often the result of early season waterlogging) or where weeds had not been controlled.

Herbicide injury (Groups B, C, & I) was detected in most crops during Jun/Jul inspections including one striking example of damage predisposing a crop of PBA HatTrick$^{(b)}$ at Billa Billa to PRR. Overall, herbicides caused no serious yield loss.

Disease management recommendations for 2016

Seed treatment and seed purity

Seed borne Botrytis, seed borne Ascochyta and several soil borne fungi can cause pre- and post-emergence seedling death. Irrespective of source of seed and year of production all chickpea planting seed should be treated with a registered seed dressing (Table 3). Proper coverage of the seed with an adequate rate of product is essential. Be confident of the identity and purity of your planting seed. If unsure acquire certified seed from a reputable seed merchant.

Table 3. Chickpea seed treatments

Active ingredient	Example Product	Rate	Target disease
thiabendazole 200 g/L+ thiram 360 g/L	P-Pickel T®	200 mL/100 kg seed	Seed-borne Ascochyta, Botrytis, Damping off, Fusarium
thiram 600 g/L	Thiram 600	200 mL/100 kg seed	Seed-borne Botrytis and Ascochyta, Damping off
thiram 800 g/kg	Thiragranz®	150 g/100 kg seed	Seed-borne Botrytis and Ascochyta, Damping off
metalaxyl 350 g/L	Apron® XL 350 ES	75 mL/100 kg seed	Phytophthora root rot

Ascochyta blight

Recommendations for Ascochyta were revised in May 2016 – please see related article in these proceedings

The following strategy should reduce losses from Ascochyta in 2016:

- In areas where AB was detected in 2015, spray all varieties, including PBA HatTrick$^{(b)}$ and PBA Boundary$^{(b)}$ with a registered Ascochyta fungicide prior to the first rain event after crop emergence, three weeks after emergence, or at the 3 branch stage of crop development, whichever occurs first.
- In areas where AB was NOT detected in 2015, spray all varieties with AB resistance lower than PBA HatTrick$^{(b)}$ with a registered Ascochyta fungicide prior to the first rain event after crop

emergence, three weeks after emergence, or at the 3 branch stage of crop development, whichever occurs first.

- 2-3 weeks after each rain event, monitor all crops irrespective of variety and spray if Ascochyta is detected in the crop or is found in the district on any variety.
- Ground application of fungicides is preferred. Select a nozzle such as a DG TwinJet or Turbo TwinJet that will produce no smaller than medium droplets (ASAE) and deliver the equivalent of 80–100 litres water/hectare at the desired speed.
- Where aerial application is the only option (e.g. wet weather delays) ensure the aircraft is set up properly and that contractors have had their spray patterns tested.

Botrytis grey mould, BGM

In areas outside Central Queensland, spraying for BGM is not needed in most years. However, if conditions favour the disease it will develop even though BGM was not a problem in 2015. Thus, in situations favourable to the disease (high biomass, average daily temperature 15 ^{o}C or higher, overhead irrigation in spring), a preventative spray of a registered fungicide before canopy closure, followed by another application 2 weeks later will assist in minimising BGM development in most years. If BGM is detected in a district or in an individual crop particularly during flowering or pod fill, a fungicide spray should be applied before the next rain event. None of the fungicides currently registered or under permit for the management of BGM on chickpea have eradicant activity, so their application will not eradicate established infections. Consequently, timely and thorough applications are critical.

Phytophthora root rot

Phytophthora root rot is a soil and water-borne disease, the inoculum can become established in some paddocks. Alternative Phytophthora hosts such as pasture legumes, particularly medics and lucerne must be managed to provide a clean break between chickpea crops. Damage is greatest in seasons with above average rainfall but only a single saturating rain event is needed for infection. Avoid high-risk paddocks such as those with a history of Phytophthora in chickpea, water logging or pasture legumes, particularly medics and lucerne. If considerations other than Phytophthora warrant sowing in a high-risk paddock, choose PBA HatTrick® or Yorker® and treat seed with metalaxyl. Metalaxyl can be applied in the same operation as other seed dressings providing all conditions of permits and labels are met. Metalaxyl only provides protection for about 8 weeks; crops can still become infected and die later in the season.

Further information

http://www.grdc.com.au/Resources/Factsheets/2013/05/Chickpea-disease-management and in the NSW DPI 2016 Winter Crop Variety Sowing Guide.

Acknowledgements

This research is made possible by the significant contributions of growers through both trial cooperation, paddocks access and the support of the GRDC, the authors would like to thank them for their continued support.

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Chickpeas – new Ascochyta and Botrytis grey mould advice for 2016

Kevin Moore[1] and Johanna Couchman[2]

[1] Department of Primary Industries, NSW
[2] Department of Economic Development, Jobs, Transport and Resources, VIC

Key words

chickpea, Ascochyta, Botrytis, management

GRDC code

DAN00176 Northern NSW Integrated Disease Management

Take home message

- Guidelines for managing Ascochyta and Botrytis in 2016 have been revised as a result of changes to predicted winter and spring rainfall.
- Growers are advised to take a conservative approach to Ascochyta management and use an integrated management strategy of agronomy and fungicide application in Northern Region chickpea crops (with the exception of Central Queensland).
- Have at least 2-3 Ascochyta and 1-2 Botrytis fungicides on farm.
- In most situations, apply an Ascochyta fungicide to ALL varieties (including PBA HatTrick(D) and PBA Boundary(D)) BEFORE the first post-emergent rain event.
- Be prepared to apply a BGM fungicide in early-mid September

Changes to the 2016 winter crop weather forecast

For the Northern Region the long term seasonal forecast has moved from predicted average early winter rainfall, and a probable El Niño, to above average winter rainfall combined with La Niña conditions in spring. This forecast, combined with evidence that the Ascochyta blight (AB) fungus is changing and concerns about varietal purity in the northern region, means chickpea growers will need to take a conservative approach to Ascochyta management. Mild, wet winter conditions will also produce high biomass crops and, combined with a wet spring, will favour Botrytis Grey Mould (BGM).

Reducing foliar disease risk through agronomy

Delaying planting will reduce the number of disease cycles to which the crop is exposed, however this increases the risk that it may start raining and remain too wet to plant. In this situation, planting on wider rows (75cm or greater) will provide better aeration, delayed canopy closure and improved penetration and coverage by foliar fungicides. Planting deeper will prolong emergence and achieve a similar result to delaying planting.

Be prepared – have fungicides on farm

There is a high possibility of a global shortage of chlorothalonil and mancozeb fungicides in 2016. If possible, stocking 3-4 Ascochyta sprays in high Ascochyta risk areas and 2-3 sprays in lower risk areas on farm would protect growers from such a shortage. There will also be strong demand for BGM fungicides from the lentil industry and growers are advised to have 1-2 BGM sprays available on farm. In addition, Pulse Australia has already obtained Minor Use Permits for alternative Ascochyta fungicides.

Be proactive with Ascochyta fungicide application

In the 2016 season, growers will face a few different scenarios with regard to Ascochyta management.

Irrespective of whether Ascochyta was detected in 2014 or 2015 in your district, all varieties rated Susceptible (S) (e.g. Kyabra(Ⓑ)) or Moderately Susceptible (MS) (e.g. PBA Monarch(Ⓑ)) should be treated with a registered Ascochyta fungicide before the first post emergent rain event. Central Queensland growers should consult with their agronomist.

In the following situations, it is recommended that growers spray with a registered Ascochyta fungicide BEFORE the first post emergent rain event:

- If Ascochyta was found in your district in 2014 or 2015;
- If Ascochyta was found on volunteers over the 2015/16 summer;
- If you are uncertain of purity of your variety - purity of your variety is best determined by asking yourself: How confident am I that every plant in my crop of PBA HatTrick(Ⓑ) is a HatTrick(Ⓑ) plant?
- If Ascochyta was not detected in your district in 2014 or 2015 and was not found on volunteers over 2015/16 summer, but you want to minimize your risk of Ascochyta.

If none of the above scenarios apply to your situation and you are prepared to accept some risk of Ascochyta, wait until Ascochyta is detected before activating a fungicide program. It should be noted that a lack of detection of Ascochyta in your crop or district does not mean it is not present. There have been several cases where Ascochyta was not detected in a previous crop, as was the case in 2014 and 2015, but became widespread on a subsequent crop or on volunteers.

Botrytis Grey Mould (BGM)

Unlike Ascochyta, if conditions favour BGM in 2016 it will occur irrespective of what has happened earlier in the season, including the use of Ascochyta fungicides. If the canopy is likely to close by mid to late September, apply a registered BGM fungicide. Consult your agronomist as to whether to apply a second BGM spray.

Acknowledgements

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Effect of chickpea ascochyta on yield of current varieties and advanced breeding lines – the 2015 Tamworth trial VMP15

Kevin Moore, Kristy Hobson, Steve Harden, Paul Nash, Gail Chiplin and Sean Bithell, Department of Primary Industries, Tamworth, NSW

Key words

Ascochyta, variety, management

GRDC code

DAN00176, DAN00151

Take home message

- Under extreme disease pressure, Ascochyta can be successfully and economically managed on susceptible varieties such as Kyabra(b) and Jimbour(b).
- However, Ascochyta management is easier and more cost effective on varieties with improved resistance eg PBA HatTrick(b) and PBA Boundary(b)
- The 2015 Ascochyta trial, VMP15, confirmed the next variety planned for release (CICA0912) has excellent resistance to Ascochyta

2015 Tamworth Ascochyta management trial, VMP15

This trial sought to match Ascochyta blight (AB) management to a chickpea genotype's Ascochyta rating using ten varieties/advanced breeding lines with a range of Ascochyta resistance ratings: seven desis Kyabra(b) (S, susceptible), PBA HatTrick(b) (MR, moderately resistant), PBA Boundary(b) (MR), CICA0912 (putatively R, resistant), CICA1007 (putatively MR), CICA1302 (for CQ, putatively MR) and CICA1303 (for CQ, putatively MR) plus the kabulis Genesis Kalkee™ (rated MS), PBA Monarch(b) (MS, moderately susceptible) and Genesis 425™ (rated R).

There were three treatments: a regular fungicide application with regular applications of 1.0L/ha chlorothalonil (720g/L active), an alternative application variety management package (VMP) treatment with a low and off label rate of chlorothalonil; and a nil application; irrespective of treatment, all fungicides were applied before rain. Data for full rate and nil fungicide treatments only, are reported here (Table 1) because of restrictions on publishing off label results.

The trial was sown into standing cereal stubble on 18-19 May 2015 using tyne openers on 50cm row spacing in plots 4m wide by 10m long. VMP15 was split across two experiments, one on red soil, one on heavy black soil, the later had waterlogging problems which affected AB resistance (data not presented), data presented here are results for the trial on the red soil. We have seen examples of this in commercial crops of PBA HatTrick(b) eg at Yallaroi in 2014 and Gulargambone in 2015 where waterlogging stress lead to a decline in AB resistance. On 16 Jun, when plants were at the 3 leaf stage, the trial was inoculated during a rainfall event with a cocktail of 20 isolates of Ascochyta collected from commercial chickpea crops (1999-2014) at a rate of 1,066,666 spores per mL in 200L/ha water. This early and heavy rate of inoculation combined with extremely favourable conditions resulted in high levels of Ascochyta disease, so much so that the unprotected susceptible varieties were dead by the end of July and even unprotected PBA HatTrick(b) had severe damage (stem breakage). From inoculation to desiccation (1 Dec), the trial received 341mm in 46 days (32 days >1.0mm).

The first Group S VMP spray for Kyabra(b) was applied before inoculation. The first Group MS VMP spray for Genesis Kalkee™, PBA Monarch(b), CICA1302 and CICA1303 was applied after three

infection events (6 rain days, 67 mm rain since inoculation), for Group MR VMP spray (PBA HatTrick® and PBA Boundary®, CICA1007) and R (CICA0912, Genesis 425™) the first spray occurred after four infection events (14 rain days, 79 mm rain since inoculation). The number of rain days, rainfall and spray applications are summarised in Table 1.

Key findings of VMP15 (see Table 2) were:

- Under extreme disease pressure, Ascochyta can be successfully managed on susceptible varieties with frequent applications of registered rates of chlorothalonil
- Well managed Kyabra® yielded 1862 kg/ha with a GM of $954/ha
- Under extreme disease pressure, unsprayed PBA HatTrick® yielded only 417 kg/ha (GM -$4/ha)
- The new line CICA0912 performed well, yielding 1568 kg/ha (GM $844/ha) with no foliar fungicide

The performance of PBA HatTrick® in VMP15 was both a surprise and a disappointment. In all previous VMP trials at Tamworth, unsprayed (Nil treatment) PBA HatTrick® has produced substantial and profitable yields. For example in the 2010 trial, VMP10, it produced 1707 kg/ha (Table 3). 2010 also had above average rain in Jun/Jul that persisted throughout the season, so was in fact more conducive to Ascochyta than 2015 (although 2015 had more rain days in Jun/Jul than 2010).

VMP10 was sown 19 May 2010 using disc openers on 38cm row spacing in plots 4m wide by 10m long. There were four replicates (Table 3). On 17 Jun, when plants were at the 3 leaf stage, the trial was inoculated during a rainfall event with a cocktail of nine isolates of Ascochyta collected from commercial chickpea crops in 2008 and 2009 at a rate of 1 million spores per mL in 200L/ha water. From inoculation to desiccation (28 Nov), the trial received 430mm rain in 67 rain days (46 days >1.0mm) ie wetter than VMP15 both in total mm and number of rain days. Both VMP15 and VMP10 were in seasons that had regular rainfall and so supported the Ascochyta development consistently over the season and so provide a strong evaluation of current varieties and advanced breeding lines. A number of the key findings of VMP10 were similar to VMP15:

- Under extreme disease pressure, Ascochyta can be successfully managed on susceptible varieties with registered rates of chlorothalonil
- Well managed Jimbour® yielded nearly 3t/ha with a GM of $750/ha
- The performance of varieties and advanced breeding lines with improved resistance to Ascochyta provided the best gross margins

The findings below contrasted between the two VMP experiments

- In 2010 PBA Boundary® performed exceptionally well, yielding over 2t/ha without any foliar fungicide, a minimal yield loss (4%), compared with 53 % in 2015.
- Under extreme disease pressure in 2010 unsprayed HatTrick® still gave a profitable yield, but unsprayed HatTrick® yields were lower in 2015 and was not profitable

Table 1. VMP15 2015 dates, number of rain days (>1 mm rain), mm of rain and dates and number of 1 L/ha chlorothalonil applications, trial sown 18-19 May.

Date	No. days	mm Rain	1L spray
28-31 May	4	31	
12 Jun			Ist All genotypes
16*-19 Jun	4	61	
22 Jun	1	1	
30 Jun-01 Jul	2	4	
9 Jul			2nd All genotypes
10-17 Jul	8	12	
21 Jul			3rd All genotypes
24-27 Jul	4	13	
21 Aug			4th All genotypes
23-24 Aug	2	40	
1 Sep			5th All genotypes
3 Sep	1	11	
4 Sep	1	6	
16 Sep	1	4	
11 Oct			6th All genotypes
14 Oct	1	16	
22 Oct	1	18	
23 Oct	1	12	
26 Oct	1	10	7th All genotypes

*trial was inoculated with Ascochyta on 16 June 2015

The following factors in VMP15 may have contributed to the nil PBA HatTrick(b) treatment having a poorer yield (Table 2) than in previous VMP trials (Table 3):

(a) parts of VMP15 were waterlogged during Jun/Jul; we know from past experience and commercial crops that any stress including waterlogging compromises PBA HatTrick's(b) moderate resistance to Ascochyta.

(b) interaction between herbicide damage and Ascochyta resistance – VMP15 sustained minor herbicide injury in August. This may have also compromised PBA HatTrick's(b) moderate resistance to Ascochyta.

(c) change in the pathogen; the isolates used in VMP10 were collected from crops in 2008 and 2009 compared to the isolates used in VMP15 which were collected from 1999 to 2014. Recently collected isolates have shown a higher level of agressiveness on PBA HatTrick(b). See Ascochyta Variability GRDC Update paper for further information.

Table 2. Number and rate/ha of chlorothalonil sprays, cost of spraying, grain yield, and gross margin for seven desi and three kabuli chickpea varieties on red soil in the Tamworth VMP15 trial. (GMs also take into account other production costs estimated at $300/ha; chickpea price desi $730/t; kabuli $1000/t) Yield P<0.001, LSD 417kg/ha; GM P<0.001, LSD $354/ha

Variety	Rate of chlorothalonil	No. Sprays	Cost $/ha	Yield kg/ha	GM $/ha
CICA0912	1.0L	7	105	1853	984
Genesis425	1.0L	7	105	1875	1470
CICA1007	1.0L	7	105	1846	982
PBA Boundary(b)	1.0L	7	105	1755	876
PBA Monarch(b)	1.0L	7	105	1274	869
PBA HatTrick(b)	1.0L	7	105	1722	852
CICA1302	1.0L	7	105	1864	954
CICA1303	1.0L	7	105	1949	1018
Kyabra(b)	1.0L	7	105	1862	954
Kalkee	1.0L	7	105	1659	1254
CICA0912	Nil	0	0	1568	844
Genesis425	Nil	0	0	1144	844
CICA1007	Nil	0	0	1083	491
PBA Boundary(b)	Nil	0	0	1233	600
PBA Monarch(b)	Nil	0	0	887	587
PBA HatTrick(b)	Nil	0	0	417	4
CICA1302	Nil	0	0	0	-300
CICA1303	Nil	0	0	0	-300
Kyabra(b)	Nil	0	0	0	-300
Kalkee	Nil	0	0	1589	1289

Table 3. Number and rate/ha of chlorothalonil sprays, cost of spraying, grain yield, and gross margin for four desi chickpea varieties in the Tamworth VMP10 trial. (GMs also take into account other production costs estimated at $300/ha; chickpea price $450/t).

Variety	Rate of chlorothalonil	No. Sprays	Cost $/ha	Yield kg/ha	GM $/ha
Jimbour	1.0L	14	294	2988	750
[a]Kyabra(b)	1.0L	14	294	2549	553
PBA HatTrick(b)	1.0L	14	294	2604	578
PBA Boundary(b)	1.0L	14	294	2410	491
Jimbour	Nil	0	0	0	–300
Kyabra(b)	Nil	0	0	0	–300
PBA HatTrick(b)	Nil	0	0	1707	468
PBA Boundary(b)	Nil	0	0	2320	744

[a]Kyabra(b) 1.0L one of the four reps was severely affected by water logging which (i) compromised Ascochyta control and (ii) impacted on yield

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Chickpea on chickpea – is it worth it?

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Key words

Chickpea, Ascochyta, Phytophthora, Sclerotinia, management

GRDC codes

DAN00176, DAN00151

Take home message:

Planting your 2016 chickpea crop into paddocks that had chickpeas in 2015, or earlier, is risky and you could lose money.

Further, it puts current disease management practices under pressure and could lead to reduced life of chickpea varieties, development of fungicide resistance and problems with weeds and insects.

Growers are urged to follow recommendations for current best practice especially with regard to crop rotation.

Background

Tempting as they are, current chickpea prices should not lure growers into thinking back to back chickpea is a viable option. Why not? For growers, the biggest risk is you stand to lose money – a lot of money. For the chickpea industry, the concern is that current best practices will become redundant prematurely or will fail completely.

What are the risks of back to back chickpea?

The main risks are seed borne, stubble borne and soil borne diseases. Successful disease management in chickpeas relies heavily on an integrated management package involving paddock selection (crop sequencing), variety choice, seed treatment, strategic fungicide use and hygiene.

Back to back chickpea - which diseases are of concern? There are four major chickpea diseases that will be favoured by planting chickpea on chickpea, ie:

- Ascochyta blight (AB, *Phoma rabiei* – previously called *Ascochyta rabiei*)
- Phytophthora root rot (PRR; *Phytophthora medicaginis*)
- Sclerotinia rot ("Sclero" *Sclerotinia sclerotiorum* and *S. minor*)
- Root lesion nematode (RLN, *Pratylenchus* spp)

Of these, Ascochyta, Phytophthora and Sclerotinia have the potential to cause 100% loss if conditions are conducive.

The risks of Botrytis grey mould (BGM, *Botrytis cinerea*), Botrytis seedling disease (BSD, *B. cinerea*) and viruses (several species) are unlikely to increase with chickpea on chickpea UNLESS some consequence of back to back chickpea favours these diseases eg patchy, uneven stands caused by Ascochyta, Sclerotinia or Phytophthora will increase the risk of virus.

If I did not find any disease in my 2015 crop, is it safe to plant chickpea on chickpea in 2016?

The short answer is NO. Severe disease can occur even if disease was not detected in the 2015 crop or even in earlier chickpea crops. This was demonstrated clearly in 2015 in north western NSW/southern QLD.

Case 1: The bulk of one paddock had been planted in 2013 to PBA HatTrick(Φ) but a narrow strip was sown with the new variety PBA Boundary(Φ). The soil was a clay grey vertosol conducive to Phytophthora root rot when wet. PBA HatTrick(Φ) has some resistance to Phytophthora (rated MR) but PBA Boundary(Φ) is susceptible. In 2013, no Phytophthora was observed in either variety. The entire paddock grew wheat in 2014 and in 2015 was sown to PBA HatTrick(Φ). On 2 September 2015, Phytophthora (confirmed by lab test) was obvious in the area sown to PBA Boundary(Φ) in 2013 but was not detected in the bulk of the paddock sown to PBA HatTrick(Φ) in 2013. The 2015 Phytophthora was so severe in the 2013 PBA Boundary(Φ) strip that it was not harvested whereas the 2013 PBA HatTrick(Φ) area went over 2t/ha.

Case 2: In 2014 several paddocks on one farm were planted to Kyabra(Φ) (susceptible to Ascochyta blight). Ascochyta was not detected in 2014 either on the farm or in the district. This, together with the prediction of an El Nino kicking in towards the end of July 2015, led to a decision to plant Kyabra(Φ) in the paddocks that had Kyabra(Φ) in 2014. It was reasoned that if Ascochyta did occur in 2015, it could be controlled with fungicides. What was not considered would be how to manage Ascochyta if it was too wet to spray – which unfortunately is what happened in early winter. Even though no Ascochyta was detected in 2014, the pathogen was clearly on farm and infected plants in late autumn/early winter. The first fungicide was not applied until 14 July by which time the disease was well established. When inspected on 29 July 2015, Ascochyta was rampant in all paddocks and was especially severe in those that had chickpeas in 2014, with many areas of dead and stunted plants. Although no rain fell after end July, these "bad" areas only went 0.6 – 0.8 t/ha compared with Kyabra(Φ) planted into wheat stubble that went 1.0 – 1.5 t/ha.

What are the impacts of back to back chickpea on a grower?

The main short term one is losing money both from lost yield and quality and, for those diseases that can be controlled in-crop eg Ascochyta, increased production costs. Longer term consequences include increasing inoculum loads in paddocks, rendering them less productive and less flexible. For example with *Sclerotinia* spp, which have wide host ranges (including cotton), the survival structures (sclerotia) remain viable in soil for many years. Thus any practice that increases the sclerotial load reduces the potential of the paddock for host crops such as faba bean, canola, lupin, field pea, cotton (and future chickpea crops).

What are the impacts of back to back chickpea on the industry?

There are three:

1. Increased risk of changes in the pathogen ie it becomes more virulent and aggressive
2. Reduced commercial life of varieties ie back to back chickpea increases the risk of the pathogen establishing in the crop early which increases the potential for more disease cycles throughout the growing season which means resistance genes are subjected to more challenges by the pathogen. Resistance genes are limited; the loss of any gene will severely hinder the development of new chickpea varieties.
3. Increased risk of pathogens developing resistance to fungicides ie reduced life of fungicide. For diseases that can be managed with in-crop fungicides eg Ascochyta, the earlier the disease establishes, the more likely is the need for repeated applications of fungicides. If you wanted to find resistance to chlorothalonil in the Ascochyta pathogen, a good place to look would be in early sown back to back Kyabra(Φ). The problem here is that any isolate that is resistant to chlorothalonil is unlikely to be confined to the paddock (or farm) in which that resistance developed. Thus an Ascochyta isolate with resistance to chlorothalonil on a single farm in say Moree could become established in the Darling Downs and elsewhere in northern and north central NSW within a few seasons. This would be the end of chlorothalonil as a disease management tool for chickpeas.

Planting 2016 chickpeas into 2015 chickpea paddocks – is it worth it?

Definitely NOT. Besides it is doesn't make sense. As well as increased risk of disease, weed and insect management will also be more challenging. At $800/t, surely growers should be doing everything to reduce risk and maximise yield and quality.

Further information on chickpea disease management can be found at the following:

http://www.grdc.com.au/Resources/Factsheets/2013/05/Chickpea-disease-management and in the NSW DPI 2016 Winter Crop Variety Sowing Guide

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Chickpea Ascochyta – latest research on variability and implications for management

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Key words

chickpea, Ascochyta, pathogenicity, latent period

GRDC code

DAN00176, UM00052, DAN00151, DAV00126, DAN00151, DAV00098

Take home message

- In 2015, Ascochyta blight occurred in a higher proportion of chickpea crops (60 of 243 crop inspections) than in 2014 (62 of 332 crop inspections). Most infected crops were PBA HatTrick(b) which was also the most commonly grown variety.
- Work to determine if the Ascochyta pathogen is changing started in 2013, where a number of projects are working together to provide an integrated approach to chickpea Ascochyta blight to improve variety resistance and best management practices.
- Initial results show that the population varies in time for spore germination, germ tube length, ability to cause disease (pathogenicity), and time to develop fruiting bodies (latent period).
- Significant differences in the reaction of some varieties and advanced breeding lines to two aggressive isolates of the AB pathogen have been found
- It is essential that growers adhere to best management practices, such as sustainable rotations, to minimise selection pressure on the pathogen and maximise the longevity of variety resistance.
- While research into variability of the AB pathogen continues, it seems prudent to adopt a conservative approach to AB management

Ascochyta blight in 2015 chickpea crops

In 2015, 243 chickpea crop inspections were conducted as part of DAN00176. Ascochyta blight (AB) (*Phoma rabiei* formerly called *Ascochyta rabiei*) was detected in 60 crops. Inoculum had carried over from the 2014 season and wet conditions during Jun/Jul favoured infection and disease development. High chickpea prices tempted some growers to break best practice eg plant back to back chickpeas resulting in severe disease. Some growers reported more AB in PBA HatTrick(b) than they ever saw in Jimbour but many of these crops had been inundated in Jun/Jul and we know that AB resistance of waterlogged chickpeas is compromised. Further the genetic purity of the variety could not be determined. Generally, however, good management and dry conditions through Aug – Oct kept AB under control and no major yield losses were reported.

Details of chickpea diseases and a review of the 2015 chickpea season are in another paper in these Proceedings (Chickpeas – what we learnt in 2015 and recommendations for 2016).

Latest research on variability in the Ascochyta pathogen

Is the pathogen changing? Yes, and as a population of living individuals (isolates), we should expect it to change.

Has the pathogen changed in response to selection pressure such as the widespread cultivation of varieties with improved resistance or other factors? We don't yet know. To know if something has changed, you need to track it over a suitable time period. Detailed studies on molecular variability in the AB fungus commenced in 2008 and have shown that the overall population variation hasn't changed much. However, pathogenicity studies that began in 2013 indicate that there are differences in pathogenicity among isolates and that highly pathogenic isolates are causing disease on PBA HatTrick(Þ). This paper provides key results from a range of research groups working on this combined project to better understand the chickpea AB population and its threat to the resistance sources through potential adaptation and selection.

Latent period

The incubation period is the time from infection to the appearance of symptoms. The latent period (LP) is the time from infection to the development of pycnidia (the small dark fruiting bodies that develop in the leaf and stem lesions), the LP is important because it determines how fast the disease can cycle in a crop. Determining these characteristics is thus another way of measuring variability in the pathogen population.

Three experiments were conducted in 2015. In each experiment, five isolates representing a sub-set of the pathogen population in Eastern Australia plus a 6th control isolate (obtained in 2014 from PBA HatTrick(Þ) at Yallaroi, TR6415) were evaluated in a growth cabinet (20°C/15°C 12h day/12h night) on four chickpea genotypes. There were eight replicates (pots) for each of the 24 genotype by isolate combinations. At the 3 leaf stage plants were grouped by isolate and inoculated with a conidial suspension of 100,000 conidia/mL (sprayed to run-off). Plants were examined daily for symptoms and pycnidia. The mean LP was estimated by survival analysis with the status of a pot based on whether pycnidia had or had not developed. For each genotype-isolate, the data is the last day that pycnidia had not developed.

The four genotypes, their AB rating and abbreviation are: 1) ICC3996 (rated R, coded ICC), 2) Genesis™ 090 (rated R, coded GEN), 3) PBA HatTrick(Þ) (rated MR, coded HAT), 4) Kyabra(Þ) (rated S, coded KYB).

For each experiment, LP varied significantly between some isolates and genotypes (LP range 6-8 days). Furthermore, all isolates had the shortest LP on the most susceptible entry, KYB and the longest LP on the most resistant entry, ICC or the second most resistant entry, GEN (see example findings, Figure 1). Within an experiment, no single isolate had the shortest LPs on all genotypes, we interpret this as indicating there are no clear differences among isolates in the contribution of LP to isolate aggressiveness.

These experiments complement the pathogenicity work and confirm variability does exist in the pathogen population

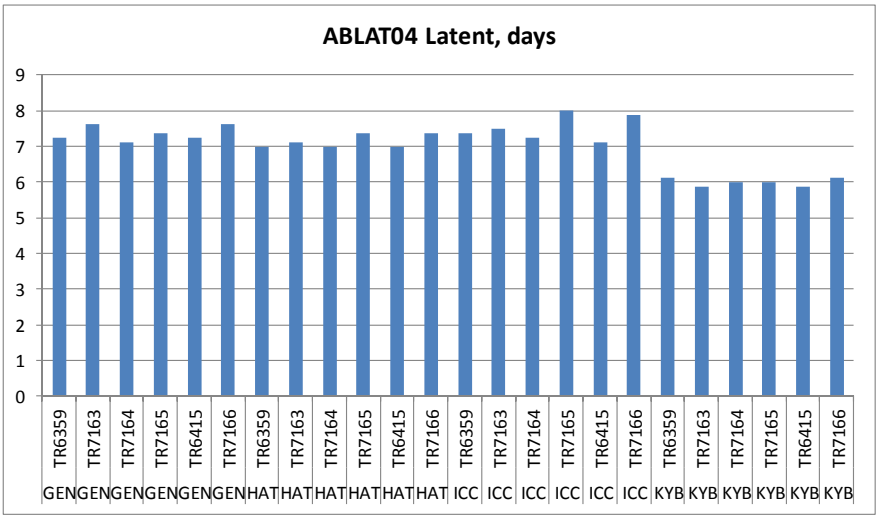

Figure1. Latent period results for experiment ABLAT04 grouped by genotype (ICC3996 (ICC), Genesis 090 (GEN), PBA HatTrick® (HAT), Kyabra® (KYB)) for inoculation with six isolates listed by isolate no, source and variety: TR6359 2014 North Star NSW, Flipper®; TR7165 2014 Horsham VIC; Genesis425, TR7163 2014 Donald VIC; Slasher®; TR6415 2014 Yallaroi NSW, HatTrick®; TR7164 2014 Donald VIC, Slasher®; TR7166 2014 Salter Springs SA, Monarch®.

Histopathology experiments

A range of preliminary histopathology experiments have been completed, see Figure 2 for summary spore germination and germ tube length results. Key findings from a range of work in this area are that:

- Spore germination begins much faster on the susceptible Kyabra® and on PBA HatTrick® than on the resistant Genesis090
- Spore germination is consistently slower and lower on the resistance source ICC3996 than on any other chickpea genotype tested
- There is significant variation in germination time among different isolates and this correlates with their level of pathogenicity
- After germination, germ tube length prior to invasion is significantly shorter on ICC3996 than any other chickpea genotype tested

These differential fungal responses may be indicative of host recognition and defence strategies, which are being further investigated.

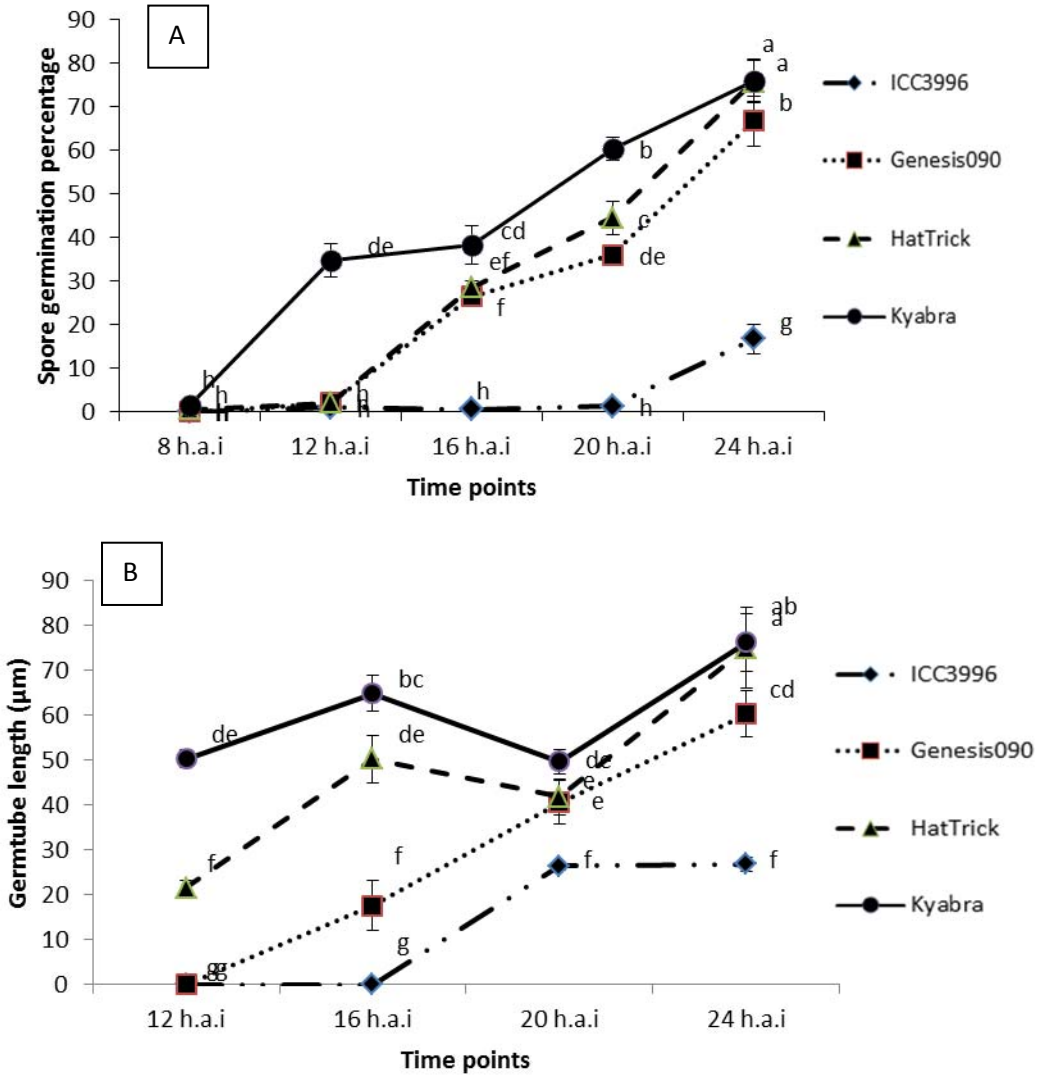

Figure 2. Significant differences were observed among the physiological traits of a highly pathogenic isolate FT13092-1 from Kingsford, SA when inoculated onto chickpea genotypes that are resistant (ICC3996 and Genesis090), moderately resistant (PBA HatTrick(Þ) or susceptible (Kyabra(Þ). Where A = the percentage of germinated spores and B = the germtube length over time after inoculation.

How is this information used by the PBA Chickpea program?

In 2014 and 2015 two aggressive isolates identified by the pathogen variability project were screened on the national Stage 3 desi and kabuli entries in a controlled environment by SARDI. In 2015 the two isolates tested were collected in 2013; FT13092-1 from South Australia on Genesis 090 and TR5919 from northern NSW (Tooraweenah) on PBA HatTrick(Þ. Of the 154 entries tested, 62 breeding lines significantly differed in their resistance (% of main stem broken) to the two isolates (subset of lines presented in Table 1). The northern isolate was found to be more aggressive than the South Australian isolate. There was no significant difference in the response of PBA HatTrick(Þ to the two isolates, but PBA Boundary(Þ, CICA0912 and CICA1007 had significantly higher disease with TR5919. Conversely, the kabuli variety Genesis Kalkee had significantly lower disease with the TR5919 isolate compared to the SA isolate. The desi CICA1521 and kabuli CICA1156 had very low levels of disease from both isolates. The 2014 research examined two isolates collected in 2010 and a much smaller number of entries 8 (out of 137) had a significantly different response to the two isolates.

To complement this information, molecular markers have been screened across the 154 entries. A total of 5 flanking molecular markers (3 SNPs and 2 SSRs) for AB resistance (resistance sources S95362 (kabuli) and ICC3996 (desi)) were identified within "DAV00098 - Molecular markers for the pulse breeding programs" led by DEDJTR, Victoria. These markers have been validated across a diverse set of chickpea lines as part of DAV00126 program. By combining the phenotypic and genotypic information, the breeding program will gain a greater understanding of the genetic resistance in each breeding line. The wider implementation of AB molecular markers across the PBA Chickpea program has identified breeding material which may contain alternative resistance genes. Research into alternative genetic resistance genes is continuing in DAV00126. The use of alternative resistance genes in the breeding program will be essential to ensure new chickpea varieties have adequate levels of AB resistance.

Table 1. Ascochyta blight ratings, response of varieties and breeding lines (% main stems broken, lsd 29.2) to two *Phoma rabiei* isolates in a controlled environment and presence/absence (+/-) of molecular marker and source of resistance.

	AB	% of main stems broken		
Name	Field rating	Isolate FT13092-1	Isolate TR5919	Marker genotype
Kyabra	S	100	100	-
PBA HatTrick	MR	0	20	+, desi
PBA Boundary	MR	35	75	+, desi
Genesis 836	MS	8	28	Not conclusive
CICA0912	R*	0	42	+, desi
CICA1007	MR*	0	50	+, desi
CICA1521	R*	0	8	+, desi
Almaz	MS	8	8	-, suggests other genes
Genesis 090	R	0	8	+, kabuli
Genesis 425	R	8	17	+, kabuli
Genesis Kalkee	MS	50	20	--, suggests other genes
PBA Monarch	MS	3	42	+, kabuli plus others
CICA1156	R*	0	0	+, kabuli

*Advanced breeding lines, putative AB rating

While research into variability of the AB pathogen continues, it seems prudent to adopt a conservative approach to AB management

Further information

http://www.grdc.com.au/Resources/Factsheets/2013/05/Chickpea-disease-management and in the NSW DPI 2016 Winter Crop Variety Sowing Guide

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Phytophthora in chickpea varieties HER15 trial –resistance and yield loss

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Key words

Phytophthora root rot, variety, risk management

GRDC code

DAN00176, DAN00151, DAQ00186, DAS00137

Take home message

- In a wet season, substantial (94%) yield losses from PRR occur in susceptible varieties such as PBA Boundary(b). Do not grow PBA Boundary(b) if you suspect a PRR risk
- Varieties with improved resistance to PRR (PBA HatTrick(b) and Yorker(b)) can also have large yield losses (68-79%) in a very heavy PRR season
- Although yield losses will occur in very heavy PRR seasons, crosses between chickpea and wild *Cicer* species such as the breeding line CICA1328 offer the best resistance to PRR
- Avoid paddocks with a history of lucerne, medics or chickpea PRR

Varietal resistance to phytophthora root rot

Phytophthora medicaginis, the cause of phytophthora root rot (PRR) of chickpea is endemic and widespread in southern QLD and northern NSW, where it carries over from season to season on infected chickpea volunteers, lucerne, native medics and as resistant structures (oospores) in the soil. Although registered for use on chickpeas, metalaxyl seed treatment is expensive, does not provide season-long protection and is not recommended. There are no in-crop control measures for PRR and reducing losses from the disease are based on avoiding risky paddocks and choosing the right variety.

Detailed information on control of PRR in chickpea is available at:
http://www.pulseaus.com.au/growing-pulses/bmp/chickpea/phytophthora-root-rot

Current commercial varieties differ in their resistance to *P. medicaginis*, with Yorker(b) and PBA HatTrick(b) having the best resistance and are rated MR (historically Yorker(b) has been slightly better than PBA HatTrick(b)), while Jimbour is MS - MR, Flipper(b) and Kyabra(b) are MS and PBA Boundary(b) has the lowest resistance (S). PBA Boundary(b) should not be grown in paddocks with a history of PRR, lucerne, medics or other known hosts such as sulla.

From 2007 to 2015 PRR resistance trials at the DAF Qld Hermitage research Facility, Warwick QLD have evaluated a range of varieties and advanced PBA breeding lines. Each year the trial is inoculated with *P. medicaginis* at planting. There are two treatments, (i) seed treatment with thiram + thiabendazole and metalaxyl and regular soil drenches with metalaxyl (Note: soil drenches with metalaxyl not currently registered) and (ii) seed treatment with thiram + thiabendazole only with no soil drenches. The first treatment has prevented infection by the PRR pathogen in all of these trials. The difference in yield between the metalaxyl-treated plots and untreated plots are used to calculate the yield loss caused by PRR i.e. % loss = 100*(Average yield of metalaxyl-treated plots – Average yield of nil metalaxyl plots)/ Average yield of metalaxyl-treated plots.

Yields in metalaxyl-treated plots were close to seasonal averages for the 2015 season with the lowest yielding breeding lines and varieties (CICA1328, Yorker(b) and PBA HatTrick(b)) yielding close to 2.5 t/ha (Table 1).

In 2015 the level of PRR in the trial was considerably higher than those previous seasons such as 2014 (Table 2). For example yield losses were greater than 40% for CICA1328 in 2015 but only 1.8% in 2015 and yield losses for PBA Boundary(b) were 94% in 2015 and 74% in 2014. However, the 2015 trial again confirmed that Yorker(b) and PBA HatTrick(b) had better resistance than PBA Boundary(b) (Table 1), which has been consistent across previous trials.

Results for the high PRR disease season of 2015 showed that susceptible varieties sustain substantial yield loss from PRR and that varieties with moderate resistance have reduced losses. The 2015 trial again confirmed the superior PRR resistance of the PBA breeding line CICA1328 which is a cross between a chickpea (*Cicer arietinum*) line and a wild *Cicer* species.

CICA1007 was included in the 2015 trial because it has high yield and large seed size in a Yorker(b) background. In the absence of PRR it was the second highest yielder in the trial (2.93t/ha) and its yield loss to PRR was similar to Yorker(b).

Table 1. Yields of commercial chickpea varieties and breeding lines protected from Phytophthora root rot, and % yield losses from PRR in a 2015 trial at Warwick QLD. (P Yield<0.001; lsd Yield = 0.46)

Variety/line[A]	Yield (t/ha) in absence of *Phytophthora* infection	Yield (t/ha) in presence of *Phytophthora* infection	% yield loss due to *Phytophthora* infection
CICA1328[A]	2.64	1.54	41.7
D06344>F3BREE2AB027[A]	2.52	1.05	58.4
PBA HatTrick(b)	2.50	0.81	67.7
Yorker(b)	2.61	0.57	78.7
CICA1007	2.93	0.71	75.9
CICA0912	2.76	0.37	86.6
PBA Boundary(b)	2.88	0.17	94.0

[A] These lines are crosses between chickpea (*C. arietinum*) and a wild *Cicer* species

Table 2. Yields of commercial chickpea varieties and breeding lines protected from Phytophthora root rot, and % yield losses from PRR in a 2014 trial at Warwick QLD. (P Yield<0.05; lsd Yield = 0.80)

Variety/line[A]	Yield (t/ha) in absence of *Phytophthora* infection	Yield (t/ha) in presence of *Phytophthora* infection	% yield loss due to *Phytophthora* infection
CICA1328[A]	2.76	2.71	1.8
Yorker(b)	3.01	2.69	10.4
CICA1211	3.01	2.66	11.6
D06344>F3BREE2AB027[A]	2.93	2.13	27.4
PBA HatTrick(b)	2.94	1.98	32.8
CICA0912	3.23	1.79	44.6
PBA Boundary(b)	2.79	0.73	73.8

[A] These lines are crosses between chickpea (*C. arietinum*) and a wild *Cicer* species

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Part I: New pathotypes of wheat leaf rust: potential impacts and what to look for and Part II: Adult plant resistance and rust management decision making

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Key words

Leaf rust, wheat, minimum disease standards, resistance, pathotype

GRDC codes

US00063, US00064, US00067

Take home messages

- Rust pathogens spread freely and rapidly through the Australasian region. While this is predominantly in a west-to east direction, recent years have seen two examples of east-to-west transport.
- Monitor for the presence of the green bridge, and if present, make sure it is destroyed at least 4 weeks before crops are sown, either by heavy grazing or herbicides.
- Warm, moist autumn conditions favour the development of leaf rust.
- Monitor crops of vulnerable varieties for leaf rust in 2016 and send samples for pathotype analysis to the Australian Rust Survey. This service is free to all, and is funded by the grower levy paid to the Grains Research and Development Corporation.
- The identification of rust pathotypes involves greenhouse tests in which seedlings of indicator varieties are infected, and takes about 3 weeks. These tests are increasingly being supplemented with DNA-tests that are much quicker (less than 48 hours). The DNA tests provide useful basic information but are nowhere near powerful enough to identify pathotypes.
- Genetic resistance to rust in cereals delivers significant benefit to Australian grain growers, estimated at $1.1 billion annually with wheat alone, and remains the basis of rust control.
- Minimum disease standards remain important for industry-wide benefit from genetic resistance.

New pathotypes of wheat leaf rust: potential impacts and what to look for

Australian wheat crops are infected by 3 different rust pathogens: stem rust (caused by *Puccinia graminis* f. sp. *tritici*), stripe rust (caused by *Puccinia striiforimis* f. sp. *tritici*), and leaf rust (caused by *Puccinia triticina*).

What is a rust pathotype?

Many people who have an interest in cereal production would have heard the term “pathotype” (pt., aka “races” or “strains”). Pathotypes are variants within a pathogen that differ in their ability to overcome rust resistance genes in cultivars. A good recent example of this concerns stripe rust and the wheat cultivar Mace(). Like many current wheat varieties grown in WA, Mace() carries the stripe rust resistance gene *Yr17*, a gene that is expressed at all growth stages (often referred to as seedling resistance genes, major resistance genes, all stage resistance genes; see below). While Mace() is resistant to the “WA stripe rust pathotype”, first detected in 2002, the resistance provided by *Yr17* was overcome in eastern Australia by a new pathotype, 134 E16 A+ Yr17+, first detected in 2006. To date, the latter Mace()-virulent pathotype has not been detected in WA. For this reason Mace() is regarded as susceptible to stripe rust in eastern Australia, and resistant to stripe rust in WA.

Thirteen pathotypes of wheat leaf rust have been detected in north eastern Australia since 2000, of which six have been common in recent years (**Table 1**).

Rust pathotype surveillance

The existence of rust pathotypes was first shown in the early 1900s in the USA. Not long after, Australian annual rust surveys were initiated at the University of Sydney, and continue to this day at the University's Plant Breeding Institute (PBI). The identification of rust pathotypes at the PBI is a free service that is open to anyone who would like to submit a sample for analysis. Directions on how to do so are provided at the end of this paper. Following this procedure is vital if the viability of a rust isolate is to be ensured.

Pathotype identification involves infecting seedlings of a set of cereal varieties, each carrying a different rust resistance gene, with a field collected sample of rust. The ability or inability of the rust isolate to overcome the resistance gene in each variety allows the pathotype or pathotypes present to be identified. These tests take about 3 to 4 weeks to complete, and if a new pathotype is suspected, often a longer time is needed to confirm this. The pathotype identification work at PBI is increasingly being supplemented by DNA profiling, which is comparatively quicker and may only take several days. However, while providing important information and a means by which exotic rust incursions can be recognised rapidly, as yet, DNA profiling is nowhere near powerful enough to identify individual pathotypes.

The long-term studies of pathogenic variability of rust pathogens conducted at PBI have clearly established that Australia and New Zealand comprise a single rust epidemiological unit, within which rusts migrate freely and rapidly. This is why a nationally coordinated approach to the genetic control of cereal rusts (i.e. the Australian Cereal Rust Control Program) is fundamental to success.

The annual surveys of rust variability carried out at PBI have and continue to form the basis of all gene-based rust control efforts. They monitor the effectiveness of rust resistance genes in commercial cultivars; determine the implications of new rust pathotypes in the rust responses of current cereal cultivars; facilitate the discovery and introduction of new resistance genes into locally adapted germplasm; and allow pre-emptive resistance breeding.

Table 1. Current common wheat leaf rust pathotypes detected in north eastern Australia

Pathotype	Year first detected	Comments
104-1,2,3,(6),(7),11	1989	Derived by mutation from pt. 104-2,3,(6),(7),11
104-1,2,3,(6),(7),11 +Lr37	2002	Derived by mutation from pt. 104-1,2,3,(6),(7),11
76-1,3,5,7,9,10,12 +Lr37	2011	Derived by mutation from pt. 76-3,5,7,9,10,12 +Lr37
76-3,5,7,9,10,12,13 +Lr37	2013	Derived by mutation from pt. 76-3,5,7,9,10,12 +Lr37
76-1,3,5,7,9,10,12,13 +Lr37	2014	Derived by mutation from pt. 76-3,5,7,9,10,12,13 +Lr37
104-1,3,4,6,7,8,10,12 +Lr37	2014	Exotic incursion, origin unknown

Recent changes in the wheat leaf rust pathogen in eastern Australia

A new pathotype of the wheat leaf rust pathogen, *Puccinia triticina*, was detected in a sample of leaf rust collected from a crop of the wheat cultivar SQP Revenue at South Bool Lagoon (South Australia) in mid-August 2014. The new pathotype, 104-1,3,4,6,7,8,10,12 +Lr37, was considered to be an exotic incursion based on its unique virulence profile and SSR fingerprint. This pathotype is the 12th documented incursion of an exotic wheat rust pathogen since Australia-wide cereal rust surveys conducted by University of Sydney staff began in 1922.

Following its initial detection in SA, pt. 104-1,3,4,6,7,8,10,12 +Lr37 spread rapidly throughout much of the eastern Australian wheat belt and in late September 2015 it was identified in samples of leaf rusted wheat collected from four separate locations in the northern region of the WA wheat belt.

Pt. 104-1,3,4,6,7,8,10,12 +Lr37 carries virulence for the resistance genes *Lr27+Lr31*, and the adult plant resistance (APR) gene *Lr12*, and combines this with virulence for *Lr13* and *Lr37*. All four resistances occur in Australian wheat varieties, and consequently this pathotype has resulted in increased leaf rust susceptibility in some varieties.

Of the 37 varieties for which detailed information is available, the leaf rust responses of 31 are not expected to change (**Table 2**). The remaining six carry resistance genes either singly or in combination that prior to the detection of the new pathotype would have provided some protection against leaf rust. While all of these varieties are now more susceptible to leaf rust, it is very fortunate that all except Mitch◊ and Wallup◊ carry a level of residual resistance due to the presence of uncharacterised APR. Growers of these varieties are nonetheless advised to monitor crops for the presence of leaf rust.

The leaf rust responses of the newer varieties B53◊, Buchanan◊, Flanker◊, Kiora◊ and Mansfield◊ are currently not well known, and further data will be collected during the 2016 cropping cycle.

If any rust is found on any cereal crop, it can be sent to the Australian Rust Survey (see below), where it will be analysed and the sender will be notified of the results. This is a free service, and its success in establishing the distribution and occurrence of known rust pathotypes, and in detecting new rust pathotypes, depends entirely on the collection and submission of samples.

Table 2. Leaf rust response and genotype for wheat varieties grown in north-eastern Australia[a]

Change in response due to new pathotype?	Cultivar	Leaf rust response	Rust resistance genotype	
			All Stage	Adult Plant
No	Adagio(b)	MSS	*Lr37*	Uncharacterised
No	Baxter(b)	S	*Lr17a*	***Lr34***[b]
No	Beckom(b)	S	*Lr3a, Lr37*	***Lr34***
No	Bolac(b)	S	Nil	***Lr34***
No	Cobra(b)	MR	*Lr3a,* ***Lr23***	Uncharacterised
No	Dart(b)	SVS	*Lr1, Lr13*	***Lr34***
No	EGA Gregory(b)	MR	*Lr3a,* ***Lr23***	***Lr34***
No	EGA Wedgetail(b)	MS	Nil	***Lr34***
No	EGA Wylie(b)	MS	*Lr3a, Lr17a*	***Lr34***
No	Elmore CL Plus(b)	RMR	*Lr24*	***Lr34***
No	Forrest(b)	MS	*Lr1, Lr13*	***Lr34***
No	Gauntlet(b)	MS	*Lr3a, Lr37*	***Lr34***
No	Gazelle(b)	MR	*Lr24, Lr37*	Uncharacterised
No	Impala(b)	SVS	*Lr37*	***Lr34***
No	Janz	MRMS	*Lr24*	***Lr34***
No	Lancer(b)	RMR	*Lr24*	***Lr34***
No	Livingston(b)	MSS	*Lr1, Lr13, Lr37*	***Lr34***
No	Manning(b)	MRMS	***Lr23****, Lr26, Lr37*	Uncharacterised
No	Merlin(b)	MS	*Lr1*	Uncharacterised
No	Naparoo(b)	S	*Lr13, Lr24*	Nil
No	Orion(b)	R	*Lr20, Lr37*	Uncharacterised
No	Scenario(b)	MSS	*Lr37*	Uncharacterised
No	Sentinel(b)	R	*Lr26*	***Lr34***
No	SF Ovalo(b)	MSS	*Lr13*	Uncharacterised
No	Spitfire(b)	S	*Lr1*	***Lr46***
No	SQP Revenue(b)	SVS	*Lr13, Lr37+*	Nil
No	Sunguard(b)	MR	*Lr24+*	***Lr34***
No	Sunvale(b)	S	*Lr37*	***Lr34***
No	Sunzell(b)	MS	*Lr1, Lr13, Lr37*	***Lr46***
No	Ventura(b)	MSS	*Lr13, Lr37*	Uncharacterised
No	Viking(b)	MSS	*Lr13*	***Lr34***
Yes	Mitch(b)	SVS	*Lr13, Lr27+Lr31*	Nil
Yes	Sunlamb(b)	MRMS	*Lr37, Lr27+Lr31*	Uncharacterised
Yes	Sunmate(b)	MS	*Lr1, Lr37, Lr27+Lr31*	Uncharacterised
Yes	Suntime(b)	MS	*Lr1, Lr37, Lr27+Lr31?*	Uncharacterised
Yes	Suntop(b)	MRMS	*Lr1, Lr27+Lr31, Lr37*	Uncharacterised
Yes	Wallup(b)	SVS	*Lr13, Lr20, Lr27+ Lr31*?	Nil
?	B53(b)	S	*Lr?*	Nil
?	Buchanan(b)	MR	?	Uncharacterised
?	Flanker(b)	MRMS		***Lr34***
?	Kiora(b)	MRMS		***Lr34, Lr46***
?	Mansfield(b)	MS		Uncharacterised

[a]For full genotypes (i.e. stem rust, stripe rust and leaf rust), see Cereal Rust Report 2016 14(4) [http://sydney.edu.au/agriculture/plant_breeding_institute/cereal_rust/reports_forms.shtml]

[b]Genes in bold font are effective against common pathotypes of the leaf rust pathogen.

Pathotype surveys and rust control

To have maximum impact in disease control, surveys of pathogenic variability in rust pathogens must be closely integrated with the development and management of new wheat cultivars. Where this has been practiced, surveys have provided both information and pathogen isolates that have underpinned rust control efforts, from gene discovery to post-release management of resistance resources. Information generated by pathotype surveys has been used to devise breeding strategies, inform selection of the most relevant isolates for use in screening and breeding, define the distribution of virulence and virulence combinations, allow predictions of the effectiveness/ineffectiveness of resistance genes, and issue advance warning to growers by identifying new pathotypes that overcome the resistance of cultivars before they reach levels likely to cause significant economic damage.

Maintaining and improving current levels of rust control

It has been estimated that 50% of the cost of plant improvement involves breeding to maintain current yield and quality levels to meet the challenges of degrading growing environments and evolving pathotypes of major pathogens ("maintenance breeding"). Protecting the *ca*. $1 billion savings to the Australian wheat industry from resistance breeding and reducing the current impact of rust diseases will only be possible if resistance remains a priority in breeding programs, and if the wheat industry as a whole continues to support genetic approaches to rust control.

Adult plant resistance and rust management decision making

Many people in the cereals industry would be familiar with the expression that a variety's disease resistance has 'broken down'. This expression can be misleading because it suggests that the variety itself has changed in some way. However, the shift in a variety's response to rust is actually caused by a change in the pathogen that causes the disease. This is why monitoring rust populations for new pathotypes is critical to informing knowledge of how a variety's resistance stacks up.

The emergence of a new rust pathotype can result in a resistant variety becoming more susceptible to rust. Because this shift is often subtle, describing the change in a variety to a new rust pathotype accurately can be difficult.

Changes in a variety's response to new pathotypes are influenced by the nature and number of genes that confer resistance to the disease. Such resistance genes protect against the disease either at all growth stages, which is called all stage resistance (ASR; also referred to as 'seedling' or 'major' resistance), or at adult plant growth stages only, which is called adult plant resistance (APR; also referred to as minor gene resistance).

Genes that confer ASR usually provide very high levels of protection against rust, while those conferring APR usually provide moderate levels of protection. A variety may carry one or both gene types, resulting in different effects on resistance levels.

Where a variety only carries an ASR gene, and this is overcome by a new rust pathotype, its resistance rating may change from highly resistant to highly susceptible.

There are many examples of such changes in a variety's resistance levels – known as the 'bust' part of what is known as the 'boom and bust cycle'. One of the first examples of this shift was recorded in the Eureka wheat variety's resistance to stem rust. Eureka was highly resistant to stem rust when it was released in 1938. However, because this variety only has one ASR gene (*Sr6*) to protect it against

stem rust, it became highly susceptible to the disease when this single gene was overcome by a new rust pathotype in 1942. Similarly, the stripe rust resistance rating of Mace(ⓟ) was downgraded from highly resistant to very susceptible because it only has one ASR gene (*Yr17*), which was overcome by a new pathotype in eastern Australia. However, in other grain growing regions such as Western Australia, Mace(ⓟ) remains highly resistant to stripe rust because its single ASR gene has not been overcome.

Adding another dimension of complexity are the many wheat varieties that carry a combination of ASR and APR genes. Having both these genes means a pathotypic change can result in a slight increase in susceptibility that occurs when the ASR gene is overcome by a new pathotype, but the APR gene is still effective in providing 'back-up' resistance .

Field testing is the only reliable way to determine the levels of back-up resistance provided by the APR gene. For example, the full impact of the new wheat leaf rust pt. 104-1,3,4,6,7,8,10,12 +Lr37 will not be known until further field tests are completed this year.

While many years of painstaking genetic research has led to a sound understanding of ASR genes, intensive genetic analyses of APR genes began only about 20 years ago. Consequently, information about the APR genes in Australian wheat varieties is incomplete, and varietal information on rust response such as that which appears in the University of Sydney's Cereal Rust Update reports (see: http://sydney.edu.au/agriculture/plant_breeding_institute/cereal_rust/reports_forms.shtml) has partial information only. The rust response and rust genotype (i.e. which rust resistance genes are present) of varieties that are currently grown in north eastern Australia are provided in **Table 2**. Where a variety is rated as having useful resistance (i.e. either: R, MR, MR-MS), and does not carry an effective ASR gene, the resistance present must be due to APR. For example, from **Table 2**:

- Sentinel(ⓟ) carries the ASR gene *Lr26*, which is not effective to currently prevailing leaf rust pathotypes (in the table, the ineffectiveness of *Lr26* is indicated by "*Lr26*" not being in bold font). It does however carry the APR gene *Lr34*. This variety is rated as highly resistant to leaf rust (R), which is due to the APR.
- Gazelle(ⓟ) carries the ASR genes *Lr24* and *Lr37*, which again are not effective against currently prevailing pathotypes. This variety is rated as Moderately Resistant (MR), which must be due to APR. The genetic basis of this APR is, however, unknown ('uncharacterised').
- Note that although the variety Dart(ⓟ) carries the APR gene *Lr34*, it is rated as being highly susceptible to leaf rust (S-VS). This is because some APR genes on their own do not provide strong levels of resistance (and is why they are sometimes referred to as 'minor genes', or 'genes of minor effect').

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Killing storage pests without mercy – fumigation strategies that work

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Key words

Fumigation, grain storage, phosphine, recirculation, pressure tests

PBCRC code

PBCRC3150

Take home message

Results of trial fumigations with phosphine conducted in 1,400 t silos to test the capability of these large storages have led us to make the following conclusions:

- Recirculation greatly facilitates the distribution of gas in large silos
- Fumigation in large silos without recirculation results in much lower concentrations in the base of the silo.
- Peak concentrations of phosphine typically occur between day 4 and 6 and decline for the rest of the fumigation.
- The current pressure half-life Australian Standard (AS2628) of 5 minutes is appropriate for large silos and is vital for effective fumigation.
- Fumigations are likely to fail where there are points of gas / fresh air leaks in a silo. Pressure testing prior to fumigation is a vital step in identifying and locating gas leaks.
- Strongly phosphine resistant rusty grain beetle can only be controlled by extending fumigation time beyond the label direction (of 20 d for blankets) or by implementing active recirculation.

There are very few options available to growers to control storage pests when an insect infestation has been detected. Phosphine, sold as the solid formulation of aluminium phosphide (AlP) under trade names such as phostoxin® or fumitoxin®, is by far the most common disinfestation treatment for stored grain.

The label was first written in the 1970's for relatively small silos and other storages. A significant number of growers are now investing in large capacity (e.g. 1,500 t), flat bottom silos for storing grain on farm. We do not know whether the label directions are appropriate for these larger storages.

Coupled with this uncertainty is the development of strong phosphine resistance in the rusty grain beetle. The resistant populations of the rusty grain beetle, found at a number of sites in eastern Australia, are significantly harder to control than other pests and label rates may need to be updated.

Fan forced recirculation of gas in large silos helps to distribute phosphine and has been advised for some time. Recirculation is not a requirement on the current label but may be a cost effective way to perform better fumigations.

The aim of this trial was to answer the following questions:

- Can strongly resistant rusty grain beetle be controlled in large farm silos?
- Is the current Australian Standard (AS2628 – 5 min pressure half-life) for silo gas-tightness appropriate for large silos?

- What concentrations of phosphine are achieved under passive gas distribution and to what extent does that lengthen the time required for complete kill of insects?
- Do large silos need recirculation for effective fumigation?
- What is an acceptable recirculation air flow rate and system design for large silos?

Two silos, labelled A and B, were fumigated at label rates. The phosphine in silo A was dispersed by natural means (passive fumigation). The gas in silo B was recirculated (active fumigation) for the first five days of the fumigation. Phosphine concentrations were monitored at four centre sampling points (headspace and at 9, 5, and 1 m above the floor) and at three points around the base wall (North, 120° and 240°) of each silo. Silo A had a Pressure Half Life (PHL) of 7 minutes and 35 seconds and silo B had a PHL of 2 minutes and 10 seconds. Both silos were leaking air at the silo base entry door during the pressure tests indicating a location for potential gas loss and dilution of gas with fresh air from outside.

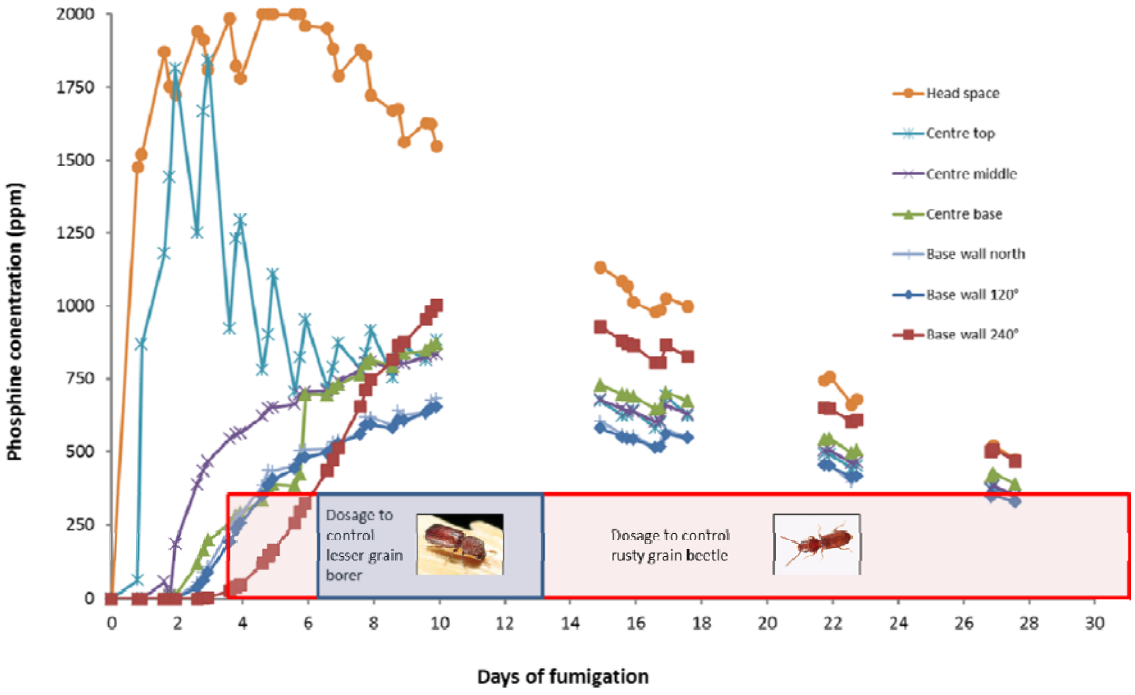

Figure 1. Phosphine concentrations measured in silo A (passive fumigation). The silo had a pressure half-life of 7 minutes and 30 seconds. The dosage (concentration x time) required to control phosphine-resistant lesser grain borer is indicted by the blue box and for phosphine-resistant rusty grain beetle by the red box.

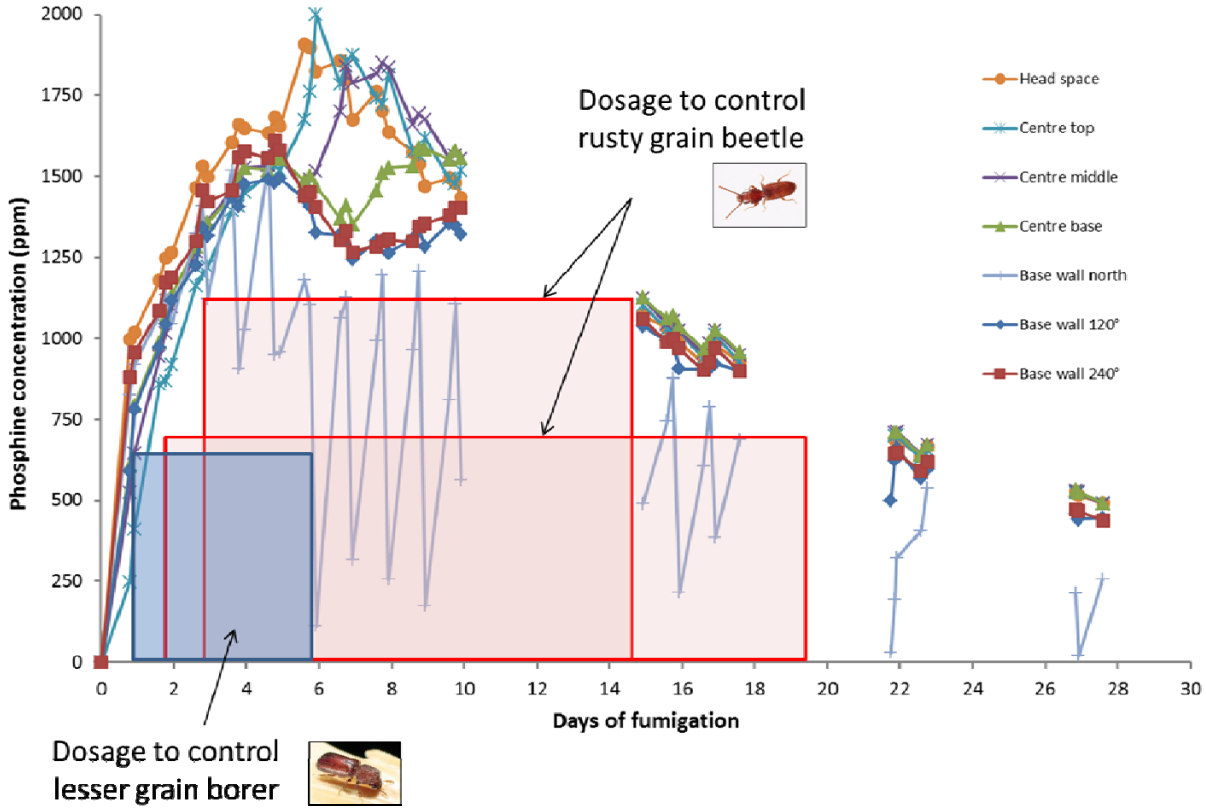

Figure 2. Phosphine concentrations measured in silo B (active fumigation). A recirculation system with an air-flow rate of 0.013 L/s/t was fitted to the silo and was run for the first five days of the fumigation. The silo had a below standard pressure half-life of 2 minutes 10 seconds. The dosage (concentration x time) required to control phosphine-resistant lesser grain borer is indicted by the blue box and for phosphine-resistant rusty grain beetle by the red boxes. Two alternative strategies to meet the required dose to control phosphine-resistant rusty grain beetle are shown. That is, a higher concentration, shorter exposure period and a lower concentration, longer exposure period.

Conclusions

- For phosphine fumigations, strongly phosphine resistant rusty grain beetle can only be controlled by extending fumigation time beyond the label direction (of 20 d for blankets) or by implementing active recirculation in gas-tight, sealable silo (AS2628)
- The current pressure half-life standard (AS2628) of 5 minutes is suitable for large silos
- Fumigation without recirculation requires a fumigation period of over 30 days
- Recirculation significantly shortened the fumigation period required to 14 days
- The label directions for solid formulations of phosphine must be updated to allow effective control of strongly resistant rusty grain beetle
- Should label rate fumigations with phosphine fail, and rusty grain beetle is identified, consider an alternative treatments such as sulfuryl fluoride (Profume®)?

Based on these conclusions, options for updating the label to ensure control of phosphine resistant rusty grain beetle include:

1. Increase application rate to maintain current fumigation period of 20 days for passive fumigations

2. Keep current application rate but extend the passive fumigation period possibly past 30 days
3. Keep the current application rate but mandate active recirculation, and maintain or possibly reduce the fumigation period
4. Increase the application rate, mandate active recirculation and reduce the fumigation period

Increasing the application rate (option 1) may be possible but would require APVMA approval and may require significant industry input to undertake residue testing etc. Increasing fumigation period (option 2) is viable but fumigations may become too long to be practical. This option is heavily reliant on silos being sealed to the Australian Standard of a 5 min pressure half-life. Mandating recirculation (option 3) would require a small capital cost to retrofit silos. Increasing the application rate in conjunction with active fumigation (option 4) could reduce fumigation times to a week or less.

A number of issues would need to be resolved if any changes are to be made to the label:

Increase application rate

- Residue testing
- WHS provisions

Increase fumigation time

- Fumigating partially filled silos
- Fumigating highly sorptive commodities such as canola

Active recirculation

- Minimum flow rates
- Fan run times

Measuring the level of silo gas-tightness

Pressure tests were carried out on silos A and B before the fumigation and at the end of the fumigation before venting, to measure silo gas-tightness. Silos were sealed and pressurised using a cordless leaf blower. Internal pressure was measured using a digital manometer (Exotech HD755) connected to the plumbing of the pressure relief valve, which comes from the headspace down the side of the silo.

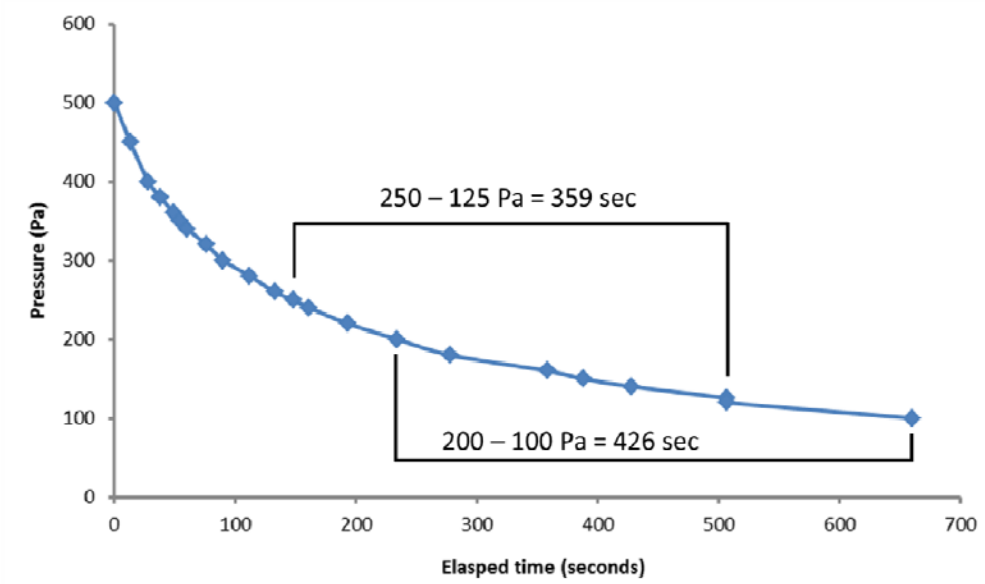

Figure 3. Pressure loss from silo A demonstrates that pressure is lost at a fast rate at higher pressures compared to lower pressures. The rate of pressure loss slows down as the pressure gets closer to atmospheric. This is why it is important to conduct pressure half-life tests using the industry AS2628 standard test method, 250 to 125 Pa.

Recirculation system fitted to silo B

A tube was connected to the pressure relief downpipe to 0.37Kw power fan (F370 Downfield, Toowoomba) positioned between the two aeration ducting trenches of the silo (Figure 4). A two way splitter was fitted to the end of a PVC pipe and two 50 mm tubes of equal length were connected to the silo aeration ducting using standard plumbing fittings. Valves (50 mm) made it possible to seal the silo at the aeration ducting and isolate the fan for removal. (The short length of white PCV pipe (ID 0.15 m) was fitted to the output side of the fan for the purpose of measuring air flows during the trial.)

Figure 4. Philip Burrill (DAF Qld) measuring air-flow in the recirculation system. For easy to follow details on how to measure air-flow in silos see http://storedgrain.com.au/testing-aeration/

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Acknowledgements

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Part I: Super cool results – achieving great aeration results and Part II: Silo recirculation as an aid to fumigation

Philip Burrill, DAF Qld.

Key words

Grain aeration, grain temperature, grain quality, storage pests, aeration fans, fumigation recirculation

GRDC code

PRB00001

Take home message

- Seek advice to ensure the right size aeration fans and associated equipment are fitted – ducting, roof vents and fan controller. Not all silo suppliers get it right.
- Recommended aeration cooling airflow rates are 2 to 4 litres of air per second, per tonne (L/s/t). Do your aeration fans achieve this when your silos are full of wheat, barley, chickpeas, sorghum, canola?
- Are you achieving the target 'grain temperatures' of 18° to 23°C during summer storage and less than 15°C during the winter period?
- Aeration maintenance: farm case studies show that aeration equipment checks and maintenance can lead to a significant improvement to aeration performance and grain storage results.
- Recirculate air with a small fan during fumigation in a sealed silo (150 – 2000 tonnes) ensures rapid, uniform distributes of phosphine gas. Otherwise it can take 2 – 5 days for gas to reach all areas inside a silo.

Storage best practice – four key steps

Aeration cooling is just one of four key best practice strategies that provide good results for on farm storage. When combined, they form the foundation for successful storage and importantly, a grower can build a reputation as a reliable supplier of quality grain.

1. Aeration: correctly designed and managed, will provide cool grain temperatures and uniform grain moisture conditions. The result is reduced problems with grain moulds and insect pests in storage, plus the ability to maintain grain quality attributes such as germination, pulse seed colour, oil quality and flour quality.
2. Hygiene: a good standard of storage facility hygiene is crucial in keeping storage pest numbers to a minimum and reducing the risk of grain contamination.
3. Monitoring: monthly checking of grain in storage for insect pests (sieving / trapping) and at the same time inspect grain quality and temperature. Keep a monthly storage record to record these details, including any grain treatments you applied.
4. Fumigation: in Australia we now only have gases (fumigation) to deal with insect pest infestations in stored grain. To achieve effective fumigations the storage/silo must be sealable – gas-tight (AS2628) to hold the gas concentration for the required time.

Effective aeration – what does it look like?

For the summer storage period November to April we aim to achieve grain temperatures of 18° to 23°C with well managed aeration cooling. For the winter period May to September the target is grain temperatures of less than 15°C.

Push a robust thermometer attached securely to a broom handle, or better, a purpose built grain temperature probe one meter into grain. Leave for a few minutes in grain before reading to see what grain temperature your aeration system has achieved.

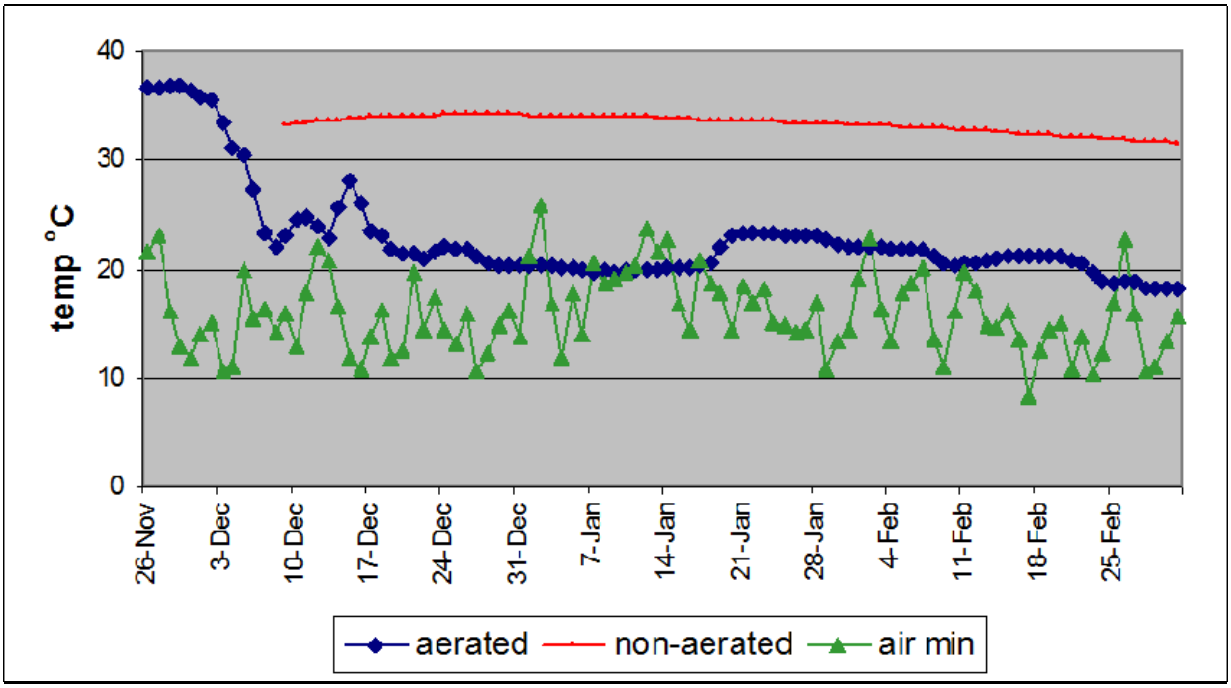

Figure 1. Two silos -wheat**.** Non-aerated silo had grain temperature sit above 30°C for 3 months, ideal for insect breeding. Well managed aeration in summer brings temperatures down towards 20°C.

Aeration - achieving good results

There are three areas to focus on for good aeration results:

a. Aeration equipment for the job

b. Operating aeration system effectively

c. Maintaining / checking the equipment is doing the job

a. Aeration equipment for the job

The three main components are fans, ducting inside the storage and the roof vents.

Fan selection: Fan size, number per silo and type of fan are common areas for confusion. It usually requires an "experienced grain aeration specialist" to provide advice to either the silo manufacturer / supplier, or directly to the grower. There are a number of important considerations to consider before fitting fans to a silo or storage.

Silo size - height & width, electricity supply available at site, grain types stored, typical harvest grain moisture contents, and what is the intended purpose of fans? Is it only for aeration cooling (2 - 4 L/s/t) , or do you want to set up one or two silos with much larger airflows (15 - 25 L/s/t) for the purpose of aeration drying?

These details can be quickly sorted out with one or two phone calls, when you are dealing with an experienced aeration specialist. It is vital that the right questions are asked. The result, the fan selection, ducting and venting design suits the intended purpose for your grain storage situation.

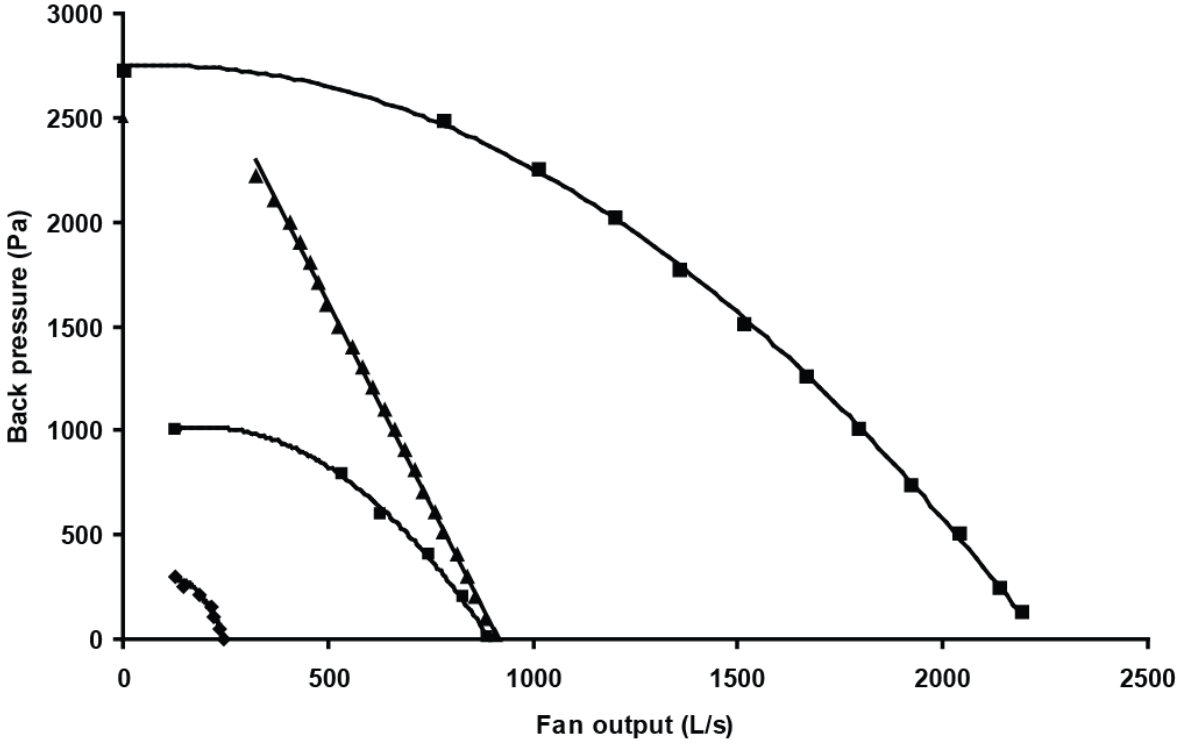

Figure 2. Note the large variation in aeration fan ouputs for four typical fans fitted to grain storages

Farm case study 1: A 130 tonne capacity cone based silo, nearly full with 105 tonnes of barley, fitted with one 0.37 kW aeration fan was tested for airflow output. Using the 'A-Flow' testing device (GRDC fact sheet, "Performance testing aeration systems") the single aeration fan was only able to generate 166 litres of air per second, or 1.6 L/s/t airflow against the 105 tonnes barley. Result: grower decided to fit a second fan (same size) on the opposite side, aiming for 3.0 L/s/t

Farm case study 2: Two Grainmaster™ 150 tonne capacity cone based silos, both fitted with a pair of 0.37 kW Agrdry F100 aeration fans. One silo was full with 140 tonnes of Soybeans and the other silo full with 150 tonnes of White French millet. With identical fans running on identical silos the total airflow output through the soybeans was 397 L/s, providing a useful 2.8 L/s/t. However airflow going into the White French millet silo was only a total of 141 L/s, providing a much lower 0.9 L/s/t. The extra back pressure on fans created by the small seed millet was reducing aeration airflow to well below the recommended cooling range of 2 – 4 L/s/t.

Ducting inside silo: There are two common types, the round tube ducting that can be made to lift up for cleaning, or the house shaped ducting that is fixed down to the cone base. Ducting length, strength, location in silo and size of perforation holes / slots, are all involved in achieving optimum airflows through grain. Ability to clean and remove grain residues from ducting for silo hygiene is important for both cone base or flat bottom silos.

Roof vents: Vents can be as simple as a "Chinaman hat style" used on the centre fill top hatch, or the many variations of "goose neck" roof vents. Unfortunately it is not uncommon to see venting design problems on range of silo brands.

The vent size / area needs to be appropriate to suit the fan output. A fan's airflow should not be used at start up to lift heavy vent lids, or constantly work against lid springs. This ensures fan airflow

is not restricted. For all sealable silos, vents require simple, effective systems for creating a gas tight seal during fumigation. Do you also have easy access to vents for maintenance on rubber seal?

Farm case study 3: Three new 150 tonne capacity, sealable, aerated silos, each fitted with two 0.37 kW Downfield F370 aeration fans (smallest curve on Fig. 2 is the F370 fan). The storage facility manager was concerned about fan output after he tested fans shortly after the silos construction was completed. He was comparing the operating sound of fans running using the four vents fitted to the roof, with the fan's sound when he also manually opened the centre top fill hatch as well. The fan performance sounded like it improved with the extra vent space provided.

When fan output was tested (A-Flow device) on the 'empty' (no grain back pressure) new silo, the pair of F370 fans could only achieve a total of 209 L/s airflow with the four vents used as designed. When the centre top fill lid was also opened, output immediately increased to 517 L/s.

On closer inspection the 4 sealable vents on the roof had no system to hold them open during aeration. There was only a long flexible cable to pull them closed / sealed for silo fumigations. Fans were losing more than half their unloaded performance, just by forcing them to lift four steel plate vent lids. Result: when the silo manufacturer was made aware of the design problem they arranged to fit a simple vent lid lifter.

Access to four vents around the roof edge to maintain rubber seals, is the next design challenge.

b. Operating aeration system effectively

Running the fan at the right times will achieve cool grain temperatures and uniform moistures. Aeration cooling aims to push through a series of 'cooling fronts' starting from the base of the silo.

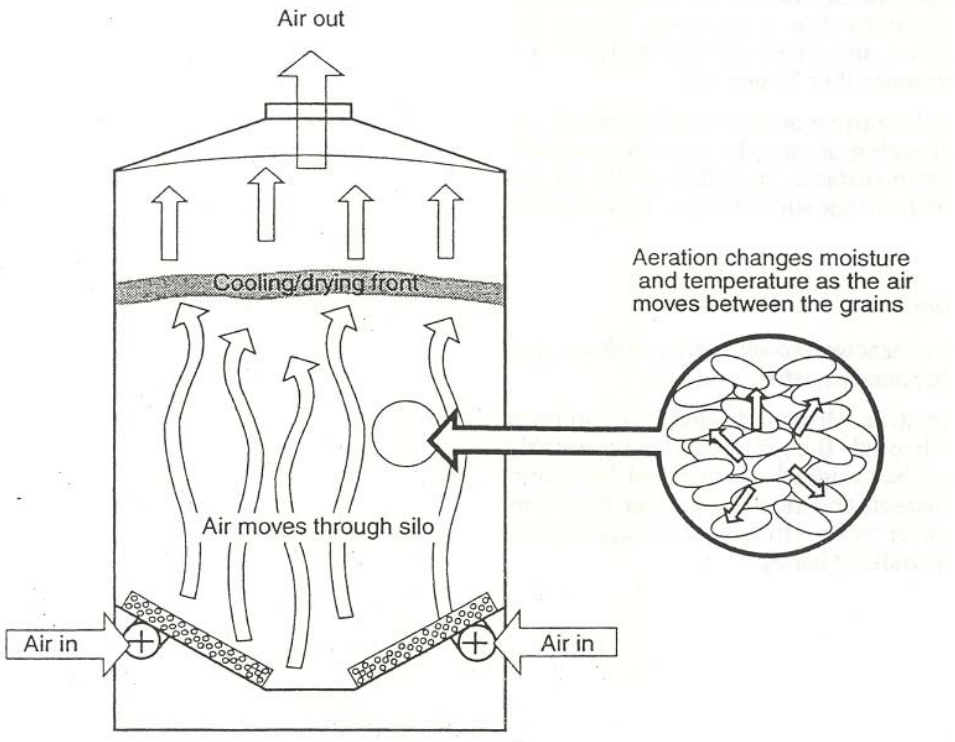

Figure 3. Cooling / drying fronts in the aeration process (C. Newman Agric. WA).

While there are a number of producers still manually operating aeration fans, for most storage facilities we recommend using a good quality automatic aeration controller with a sensor measuring both ambient air temperature and humidity to automatically turn on fans at optimum times.

Manual operation of fans

There are three stages when operating aeration cooling fans from the start of harvest:

1. As soon as enough grain covers the ducting, turn on aeration fans while filling silo. Run continuously (24hrs / day) until the first cooling front comes through the full grain depth. This usually takes 3 - 5 days. If safe, go to the top of the silo and see if the air coming out has changed from a warm, humid smell to a fresh, cool smell. The first cooling front is through. See Fig 3.
2. Once this has occurred, run the fans for approximately 12 hours per day for the next 5 – 7 days. Select the cooler night air, but avoid extended periods of high humidity air which may wet grain. Avoid fog, misty or showery conditions.
3. Check the grain temperature and condition. Grain temperature in summer should now be close to 20°C. The longer term "protect" phase now begins. Operate fan for approx. 100 hours per month, selecting cool, mostly dry air from 3 - 5 days per week to maintain cool grain conditions. An automatic controller will usually be much more reliable at this task.

Automatic controller operation of fans

Today there are automatic aeration controllers available that automatically step through the three stages outlined above.
Seek independent advice as to what are the better quality controllers to consider, as there are poor quality units that may put your stored grain at risk. Ensure the supplier has a good reputation for providing after sales support and parts if required.
For a new unit fitted to a storage facility, there is simple start up process to follow. See manual, or consult supplier. As a general rule, leave the auto controller itself powered up. It is recording a history of current weather conditions so it is able to turn fans on at the optimum times.

c. Maintaining and checking aeration equipment

There are a few basic checks and maintenance steps to ensure your system is doing the job.

1. Check grain temperatures to see if you are achieving the target temperatures of 18° to 23°C during summer storage and less than 15°C during the winter period.
2. See Fig. 4 where an OPI® cable was used in the aerated barley silo ("Farm case study 1") to record grain temperatures at various depths. This helped identify the low airflow problem.
3. When checking silos each month for insects, also look at the hour meter on the aeration auto controller to see if fans are averaging approx. 100 hours per month (+/- 20 hrs).
4. At least once per year use a good quality thermometer and relative humidity reader to check the aeration auto controller's sensor has not been damaged and is readings correctly.
5. Manually test-run fans on silos to check they are all operating. Clean fans if required.

Farm case study 4: A ten minute fan cleaning job can produce large improvements**.** A single 0.37 kW aeration fan was tested for airflow output on a 128 tonne capacity coned based silo holding 105 tonnes of barley. It was observed that the fan impeller had a significant build-up of dust on the blades prior to testing. Using the 'A-Flow' testing device, the aeration fan output was recorded as 86 L/s, or 0.8 L/s/t airflow against the 105 tonnes barley. After cleaning the dust from the blades the fan was retested and produced an output of 152 L/s, or 1.5 L/s/t. Result: grower cleaned remaining fans.

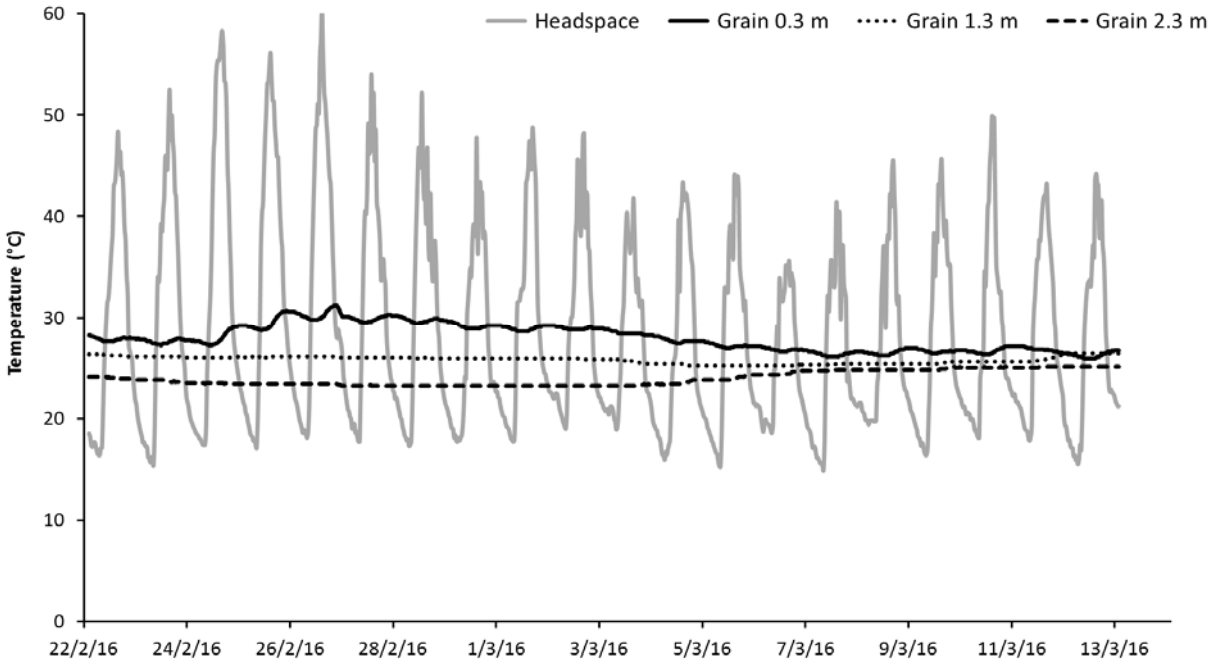

Figure 4. Temperatures in a silo of barley, in headspace and at three grain depths. The warmer than expected grain temperatures indicated possible aeration problems. See Farm Case study 1.

Silo Recirculation – how can it help with fumigation?

Australia now only has gases to control live insect pests in infested grain. Dichlorvos spray on insecticide is no longer registered for this use. To achieve effective fumigations, silos must be pressure tested to check they are sealed – gas-tight. This ensures they hold high gas concentrations for the required time to kill pests.

Silos pressure tests can be carried out by using a short burst (5 – 10 sec.) of the aeration fan, or a portable leaf blower to initially pressurise the silo for the test. The pressure decay (250 to 125 Pa) can timed by using the silo's relief valves, a length of 20 mm clear plastic tube in a "U" shape with water in it (manometer), or a digital manometer connected to the silo. See GRDC Fact Sheet : "Pressure testing sealable silos". http://storedgrain.com.au/pressure-testing/

During fumigation, phosphine gas is typically liberated over 5 or 6 days from the tablets or blankets that have been placed in the silo. This gas however only moves slowly, taking about 24 hours to travel 6 meters through grain.

If you are fumigating a medium to large silo (150 – 2000 tonne silo) the gas may take 2 – 5 days to eventually arrive in all parts of the silo. In large silo fumigations this may result in some grain, at the furthest distance from tablets, only getting 6 days of phosphine gas instead of the required 10 days or longer exposure period. Six days is not enough time to kill all the life cycle stages of the pests.

A typical phosphine fumigation required to kill all pests is a minimum of 200 ppm phosphine gas concentration for at least 10 days. See horizontal blue line in Figure 5 below.

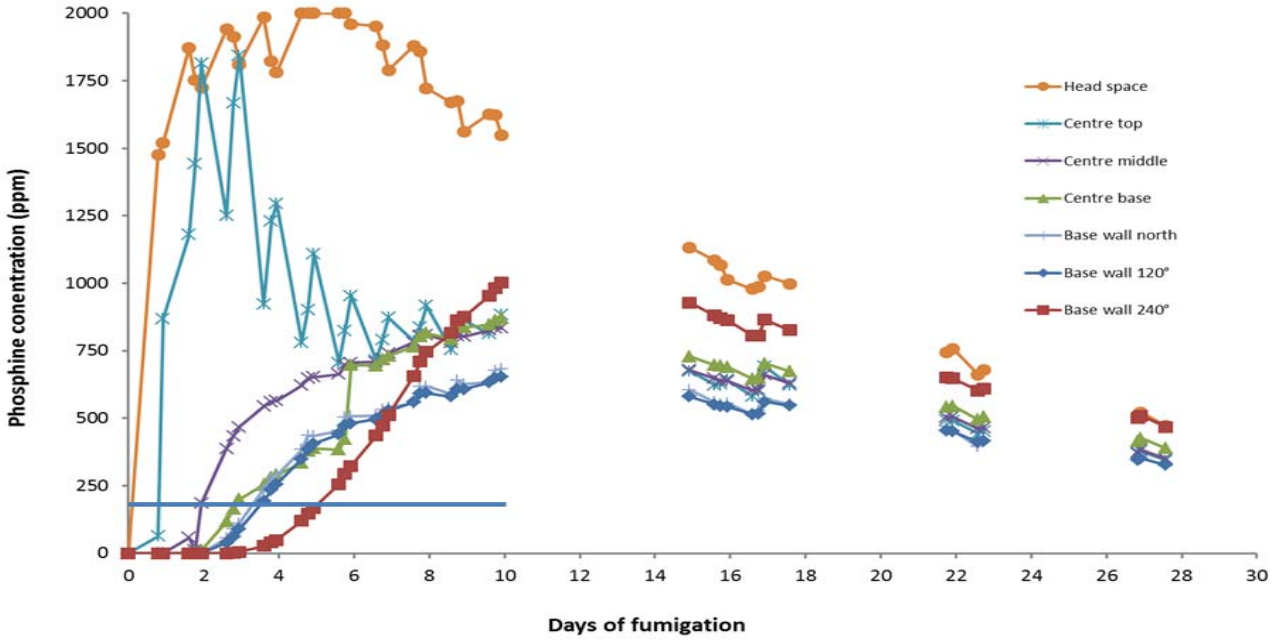

Figure 5. Phosphine gas concentrations at 7 points in a silo during fumigation of 1416 tonnes of wheat. Phosphine blankets were placed in the silo headspace with no recirculation. It took as long as 5 days for all grain at the silo base to reached at least 200 ppm gas concentration.

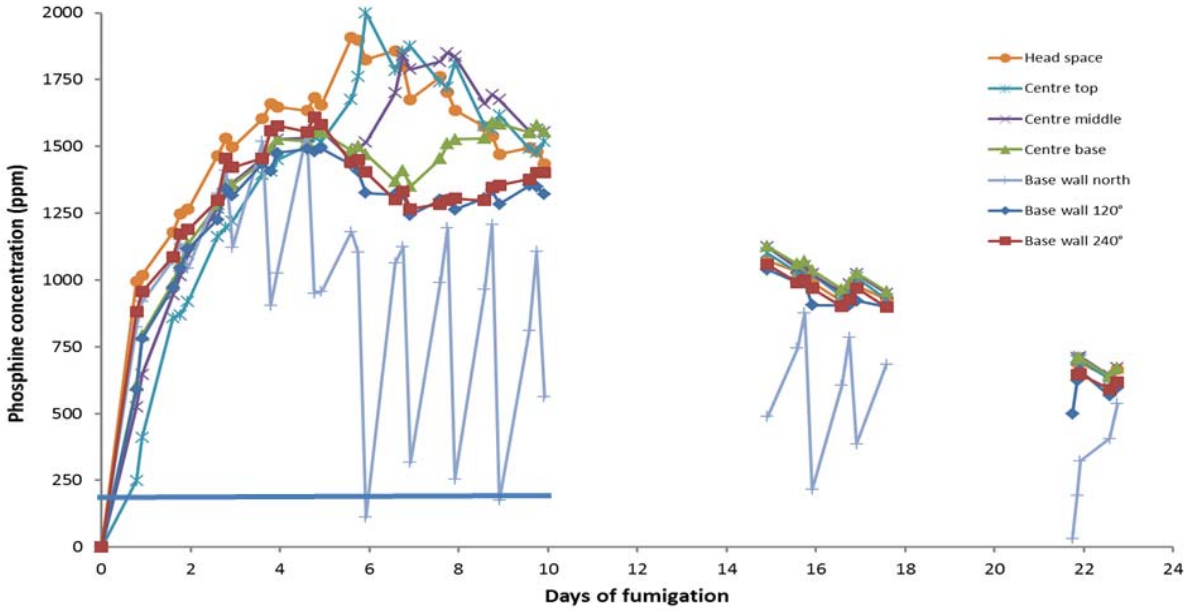

Figure 6. Phosphine gas concentations in a silo (1423 t wheat) where a small fan was used to draw gas from blankets in the silo headspace and pump it to the silo base via aeration ducts for the first 5 days of fumigation. Gas concentration in all areas of the silo reached over 800 ppm within the first 24 hrs.

Figure 7. A small fan (F370 – 0.37 kW) used during the first 5 days of fumigation to recircualte phosphine to give rapid uniform gas distribution in 1423 tonnes wheat. See Figure 6.

Options for fumigation recirculation

For all fumigation recirculation systems, the sealable silo needs to be gas – tight so there is no gas leakage during the fumigation. Fig. 6, "Base wall north" shows the impact of a leak at the silo manhole which causing large daily fluctuations in gas concentrations.

a) Phosphine blankets or tablets can be placed in the 'silo headspace' along with a small fan connected to the headspace via 90 mm pipe plumbing coming down the silo wall from the roof. Phosphine gas is drawn from the headspace and pumped into the base of the silo via both aeration ducts. See Fig. 7

b) For ground level application of tablets or blankets, a sealable 'phosphine box' can be plumbed into this system, either a moveable box, or mounted permanently on each silo.

c) Using a fan to force the phosphine gas movement around in silos during fumigation is generally recommended, rather than relying on a passive 'thermosiphon' approach. For medium and large silo fumigations, 150 tonnes plus, or silos storing smaller grain sizes (e.g. millets, canola, lentils etc.) that reduces air movement, fan force recirculation rather than thermosiphon is advised. Fan forced recirculation may also assist where the grain type (e.g. oilseeds) typically absorbs higher amounts of phosphine during fumigation.

Equipment for fumigation recirculation

- Sealable silo - gas tight, that passes pressure test
- Plumbing pipes (90 – 100 mm) from silo roof to ground level. Use quality pipe, fittings and seals that will ensure many years of safe, gas- tight fumigations
- Small fan (e.g. Downfield F370 - 0.37 kW) to recirculate air. In most case this fan size will be suitable for both small & large silos. In trials (Fig. 6 & 7) this fan size provided a complete silo air change every 12 hours for the full silo holding 1420 tonnes of wheat
- Fittings for fan intake and outlet. Flexible hoses (50 – 100mm) couplings and gate valves

Fumigation recirculation - operations

a. Pressure test the silo to check for leaks

b. Follow all label directions and place tablets / blankets in the 'headspace' or 'phosphine box'.

c. Run small recirculation fan for first 5 days of fumigation. Leave silo sealed for remaining days of fumigation expose period as label requires (e.g. 7, 10, 20 days).

   Note: There are benefits to using the silo 'headspace' to locate the blankets or tablets. The large surface area of grain in the headspace provides safe, large easy access for gas penetration and diffusion into the grain.

Warning

Always seek reliable advice before fitting fumigation recirculation systems to silos / storages. Some systems that are currently sold are not recommended because of unsafe design features. Phosphine is not only a toxic gas, but can be flammable and explosive if restricted in a small area or used in a manner that causes gas concentrations to rise quickly to high levels.

Follow label directions and seek advice.

Further reading

- GRDG Factsheet – "Performance testing aeration systems" http://storedgrain.com.au/testing-aeration/
- GRDC Fact Sheet – "Safe storage of Sunflower seed – aeration drying and cooling" http://storedgrain.com.au/safe-storage-sunflower-seed/
- GRDC Update – "How Aeration Works" http://storedgrain.com.au/how-aeration-works-grdc-update/
- GRDC booklet – Aeration stored Grain – cooling or drying for quality control http://storedgrain.com.au/aerating-stored-grain/
- GRDC booklet – Fumigating with Phosphine other fumigants and controlled atmospheres http://storedgrain.com.au/fumigating-with-phosphine-and-ca/
- GRDC video – Fumigation recirculation http://storedgrain.com.au/fumigation-recirculation/

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Russian Wheat Aphid - identification, detection, management and implications.

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Managing patches of glyphosate resistant weeds

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Tillage impacts of weed seed burial and subsequent management

Michael Widderick, Andrew McLean and Michelle Keenan, Queensland Department of Agriculture and Fisheries

Key words

Tillage, cultivation, weed seed, burial, emergence

GRDC code

UQ00062, UWA00171

Take home message

- Weed seed ecology (emergence and persistence) need to be taken into consideration when considering the potential role of tillage as a weed management tactic.
- In a zero till system, effective control of seedlings will rapidly deplete the weed seed bank of most of our key weeds. This is especially the case for feathertop Rhodes grass, windmill grass, fleabane and sowthistle.
- Herbicide resistance threatens our ability to effectively control emerged weeds.
- Applying cultivation may reduce weed emergence compared with a zero tillage system, especially in a wetter season. However, in a dry season, tillage may increase emergence.
- Seed burial is likely to increase the persistence of weed seeds.
- A second pass of cultivation can result in subsequent emergences of weeds as a result of seeds being brought closer to the soil surface.
- New approaches are being investigated to apply tillage in a targeted manner to minimise soil disturbance and weed seed burial.

Background

The widespread adoption of no-till farming has resulted in reliance on herbicides for the control of weeds. As a result of this reliance, the weed spectrum in the northern grain region has changed and become difficult to control. Herbicide resistance has become a common problem and weeds have shifted toward surface germinating species such as common sowthistle, fleabane and feathertop Rhodes grass. Continued reliance on herbicides will result in further cases of herbicide resistance and proliferation of these weeds.

The change in weed spectrum has forced industry to investigate alternative approaches for weed management, including judicious or targeted use of tillage. The challenge with the reintroduction of tillage is to retain the benefits gained through zero tillage (improved soil structure, reduced erosion and improved soil water conservation) while addressing the weed management issues mentioned above.

Tillage can be applied to target mature, uncontrolled plants or to manipulate the seedbank and thereby improve weed management. This paper primarily addresses the use of tillage to manipulate the weed seed bank.

The importance of weed seed bank ecology

Each weed species has a different seed bank ecology, including depth from which it can emerge and duration seed persist in the soil, as affected by burial depth. These ecological factors are important to understand when considering the potential role and consequences of using tillage.

Emergence

In a zero tillage system, where soil disturbance is largely removed, most weed seeds will remain in the soil surface layer (0-2cm). Germination of some of our most common weeds is favoured from these layers and helps to explain their prevalence in zero tillage systems. For example, feathertop Rhodes grass seeds emerge mostly from the top 2cm of soil with a greatly reduced emergence from 5cm (Table 1). Over 12 months, 43% of seed buried near the surface germinated, compared with 5% at 5 cm and 0% at 10cm depths.

Similarly, sowthistle will mostly emerge from the top 1cm of soil, with limited emergence from a depth of 2cm (Figure 1). Fleabane will only emerge from the top 1cm of soil, with no emergence at or below 2cm.

Other species, such as liverseed grass and barnyard grass, are able to emerge from deeper in the soil profile (Table 1). While awnless barnyard grass still prefers to emerge from a depth of 2cm, it is also capable of emerging from a depth of 5cm. Liverseed grass prefers to be buried to 5cm for optimal emergence and can even emerge from a depth of 10cm (Table 1). As a result, liverseed grass is less common in zero tillage systems.

Table 1. Cumulative emergence (% of viable seed sown) of summer grasses at 24 months following burial at different depths (cm)

Depth of burial (cm)	Feathertop Rhodes grass	Barnyard grass	Liverseed grass
0-2	43*	27	36
5	5*	5	74
10	0	0.8	16

*emergences ceased after 12 months in all studies conducted on this grass

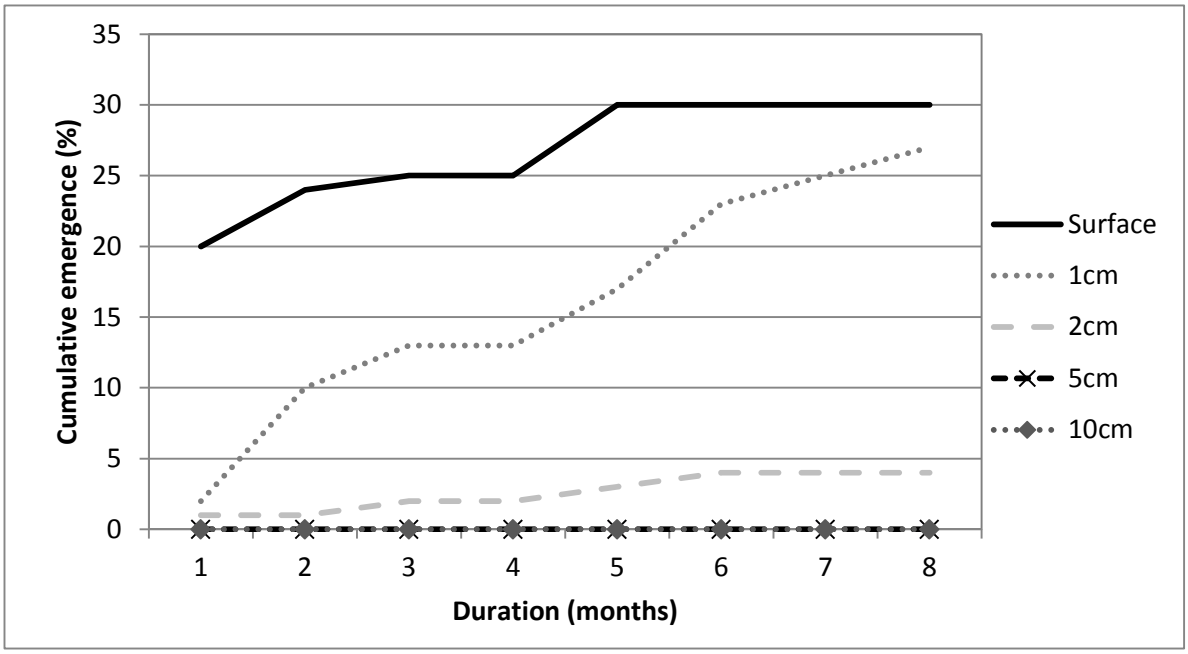

Figure 1. Cumulative emergence of common sowthistle as % of viable seed buried on the soil surface and at different depths (cm)

Persistence

Weed seeds left on or near the soil surface generally have a short life as fluctuating temperature and moisture reduces viability quickly. Generally, seed burial, for example via tillage, will promote persistence of seeds, and generally speaking, the deeper the seed is buried, the longer it will persist.

A clear example of this is barnyard grass and liverseed grass (Figure 2). For both species, seeds only remain viable for a short time in the soil surface layers, but persistence increases with depth of seed burial. Only 1 – 2% of seed remains viable after two years buried at a soil depth of 0 to 2cm, in contrast with approximately 20% remaining after two years buried at the depth of 10cm.

Depth and duration of seed burial also affect the persistence of feathertop Rhodes grass and windmill grass (Figure 2). A recent pot experiment on the eastern Darling Downs showed that after three months of burial, a large number (>70%) of FTR seed persisted at 10cm burial depth (Figure 2). There were a significantly lower number of viable seeds persisting at the 0cm depth. Burial depth had less impact on the persistence of WMG with both depth treatments having <20% viable seed remaining.

There were no further emergences of feathertop Rhodes grass from soil exhumed at 12 and 18 months showing this weed to be short lived in our southern Queensland environment. No emergences were recorded for windmill grass in soil exhumed at 18 months.

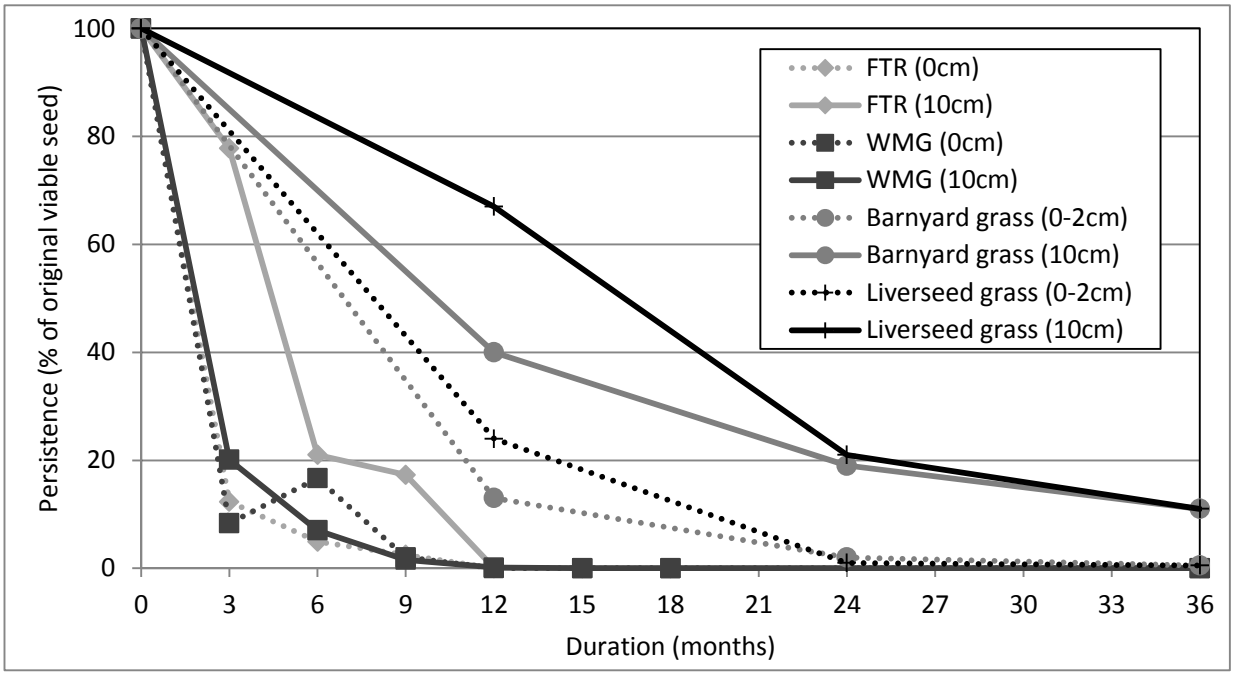

Figure 2. Persistence as assessed through emergence of seed from exhumed soil (% of viable seeds) of feathertop Rhodes grass (FTR), windmill grass (WMG), awnless barnyard grass and liverseed grass in response to different burial depths (cm) and durations of burial (months).

Even though small-seeded, both fleabane and sowthistle seed can persist at depth for more than three years. A pot study on the Darling Downs showed that after 3 years of burial 1, 10 and 8% of viable fleabane seed remained at depths of 0-2, 5 and 10cm respectively (Figure 3). For sowthistle, after 30 months of burial, over 10% of seed remained viable when buried at 5 or 10cm compared with only 1% at 1cm burial depth (Figure 4).

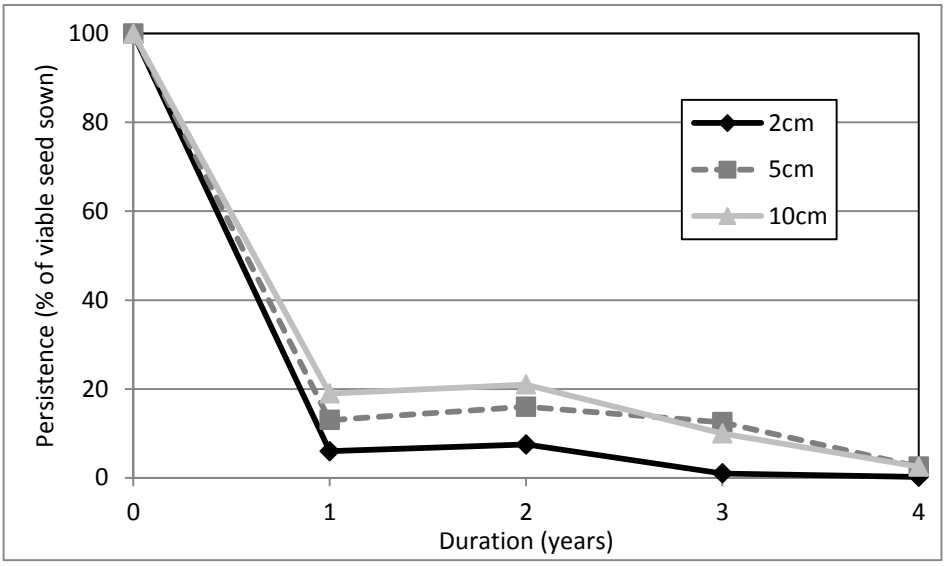

Figure 3. Persistence of flaxleaf fleabane buried at different depths and for different times (years).

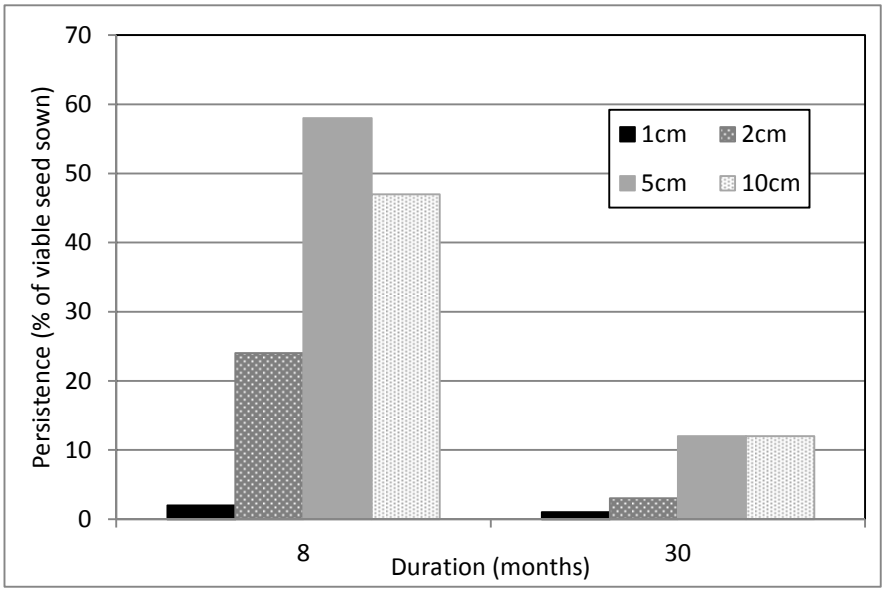

Figure 4. Persistence of common sowthistle seed buried at different depths for either 8 or 30 months.

Tillage effects

The Queensland Department of Agriculture and Fisheries (DAF) have conducted a series of four field experiments since 2011 to investigate the effect of tillage on seed burial and emergence of key weed species.

The first three experiments were established at the DAF Hermitage Research Facility, and the fourth was established at the DAF Wellcamp Research Station. At field experiments one to three, five tillage treatments were imposed with different levels of soil disturbance and inversion;

- Zero tillage (ZT)
- Harrows
- Gyral
- Offset discs
- One-way discs.

At the fourth field experiment, we explored the impact of a second tillage pass with a gyral after one-way disc and offset disc treatment, on the subsequent emergence of weeds.

Seeds of awnless barnyard grass, feathertop Rhodes grass, windmill grass, liverseed grass, common sowthistle and flaxleaf fleabane were sown on the soil surface prior to tillage application. Also, small coloured beads, to represent weed seeds, were included so we could track soil inversion via soil coring and bead recovery.

Weed emergence was counted in each treatment for up to 18 months. We were unable to get emergence of every species in each experiment therefore some of the data presented here excludes certain species.

Glass bead (seed) burial

The burial of glass beads was quite consistent across experiments. As such we are only presenting the data from the fourth experiment (Figure 5) as it also explores the impact of the second pass with the gyral.

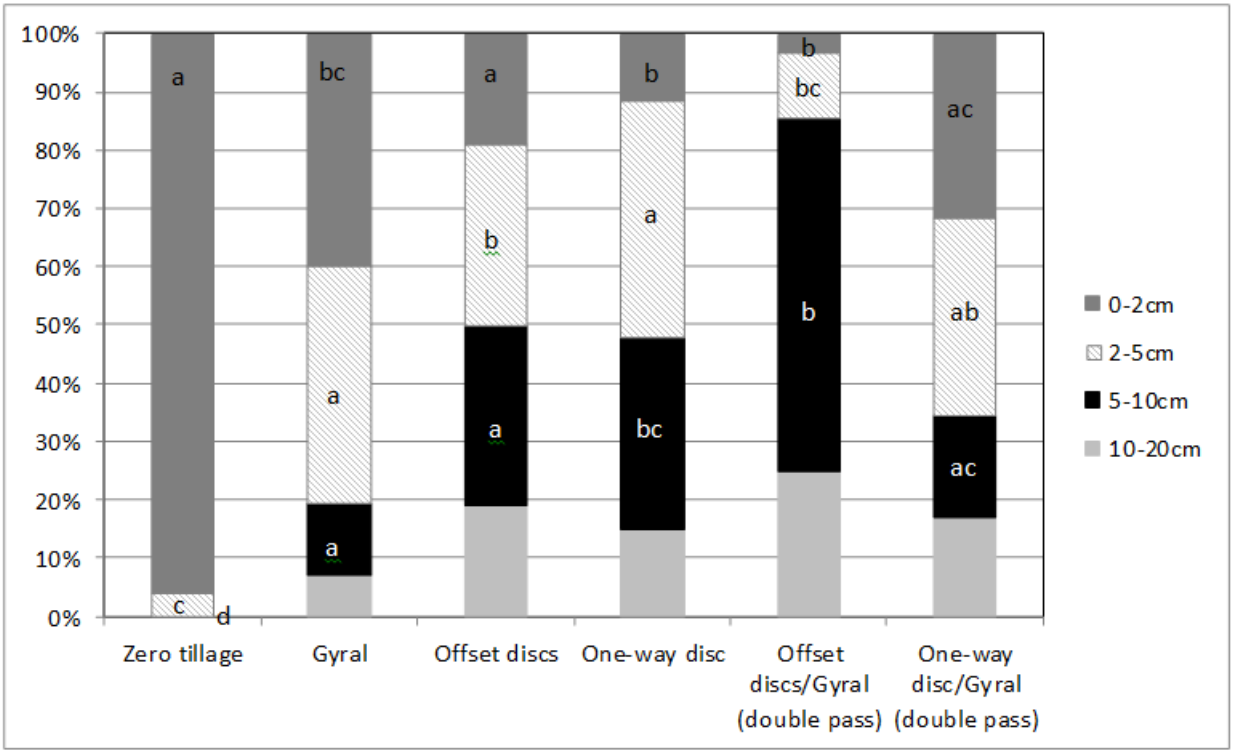

Figure 5. Burial of glass beads (cm) under different types of tillage as assessed through bead recovery from soil cores. Lettering is based upon LSD's of the transformed means. Means with the same letter within each burial depth are not significantly different at the 5% significance level.

The application of different tillage treatments affected the burial of glass beads (Figure 6). Generally, the zero tillage treatment had a larger proportion of glass beads in the top 2cm of soil and as tillage intensity increased, this proportion decreased.

Analysis showed there was a significant difference between tillage type for seed burial ($P<0.001$) at 0-2, 2-5 and 5-10cm soil depths but not for the 10-20cm depth.

For the 0-2cm depth, the offset disc treatment with and without the second gyral pass and the one-way disc treatment all significantly reduced the number of glass beads at this depth compared to zero tillage. Of note, the one-way disc double pass treatment had significantly more beads at this depth than the single one-way disc treatment showing that the second pass of the gyral returned more beads back into this layer.

For the 2-5cm depth, the zero tillage treatment had significantly fewer glass beads than all other treatments except for the offset/gyral double pass treatment. There was no significant difference between the single and double pass treatments for the offset and one-way disc treatments, showing that the second pass did not significantly alter the number of glass beads at this depth.

For the 5-10cm depth the zero tillage treatment had significantly fewer beads than all other treatments. For this depth there was a significant difference between the offset disc and the offset disc/gyral double pass treatments with a significantly greater bead count for the double pass treatment. This result shows that the double pass moved more beads into this layer.

Weed emergence

Given the difference in glass bead burial under the different tillage treatments and the impact of seed burial on emergence, it is not unexpected that we found tillage to have a significant impact on weed emergence. However, the effect of tillage on weed emergence was different across field experiments.

In this paper, we firstly report on the first field experiment to show the potential, favourable impact of tillage on reducing weed emergence (Figure 6). We will then provide an example, using sowthistle, to demonstrate how season can impact on weed emergence under different tillage treatments.

In the first experiment, cumulative weed emergence density in zero tillage treatments was 233 (fleabane), 149 (feathertop Rhodes grass), 433 (windmill grass), 267 (barnyard grass), 380 (sowthistle) and 72 (liverseed grass) plants/m^2 across a 3m^2 assessment area (to account for horizontal seed movement).

Most forms of tillage greatly reduced the emergence of all weed species (Figure 6) with the greatest reduction evident in the small-seeded species fleabane. Generally, as the intensity of tillage increased, the emergence of weeds decreased. The greatest reduction in emergence was generally under a one-way disc, which caused large amounts of soil to be inverted and seed burial.

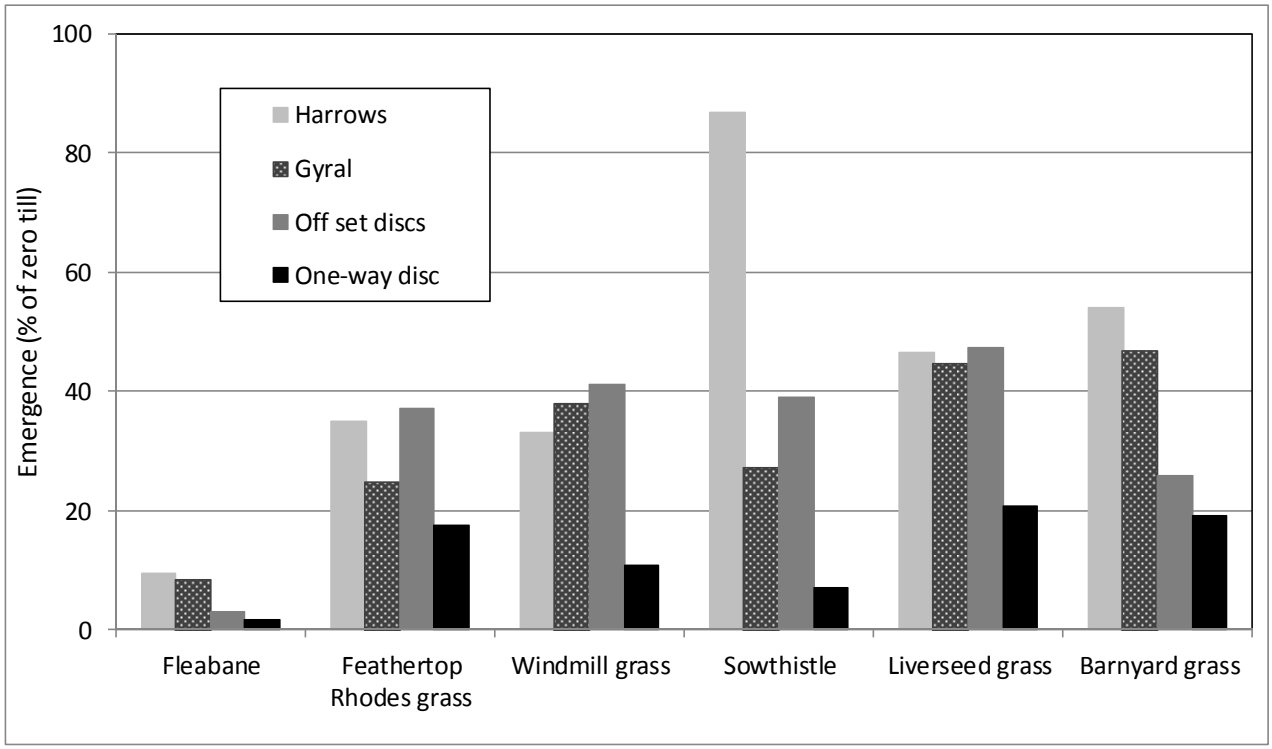

Figure 6. Emergence of key northern region weed species, as a % of emergence in zero tillage treatment, under different types of tillage

The effect of tillage on weed emergence was not consistent across experiments. For example, for sowthistle, off set discs and one-way discs reduced emergence compared with zero tillage in all four field experiments (Figure 7). However, in field experiment 2 both the harrow and gyral treatments increased seedling emergence and in experiment 3 the gyral treatment increased emergence.

This difference can be explained by considering the season (temperature and rainfall) and depth of seed burial. At the start of experiments 2 and 3, there was hot and dry weather, resulting in minimal emergence from zero tillage treatments and a rapid depletion of seed on the soil surface. In these experiments, seed buried by the harrow and gyral treatments were preserved at a depth from which they could later emerge once a favourable environment was present (sufficient moisture and suitable temperature). The emergence from the offset disc and one-way disc treatments was always less than in zero tillage treatments as the seed was buried too deep for emergence.

In experiments 1 and 4, there was a wet start to the experiment, resulting in a large flush of sowthistle emergence from zero tilled treatments. In these experiments, a portion of seed was

buried below the depth of emergence in all tilled treatments and therefore a reduction in emergence was measured.

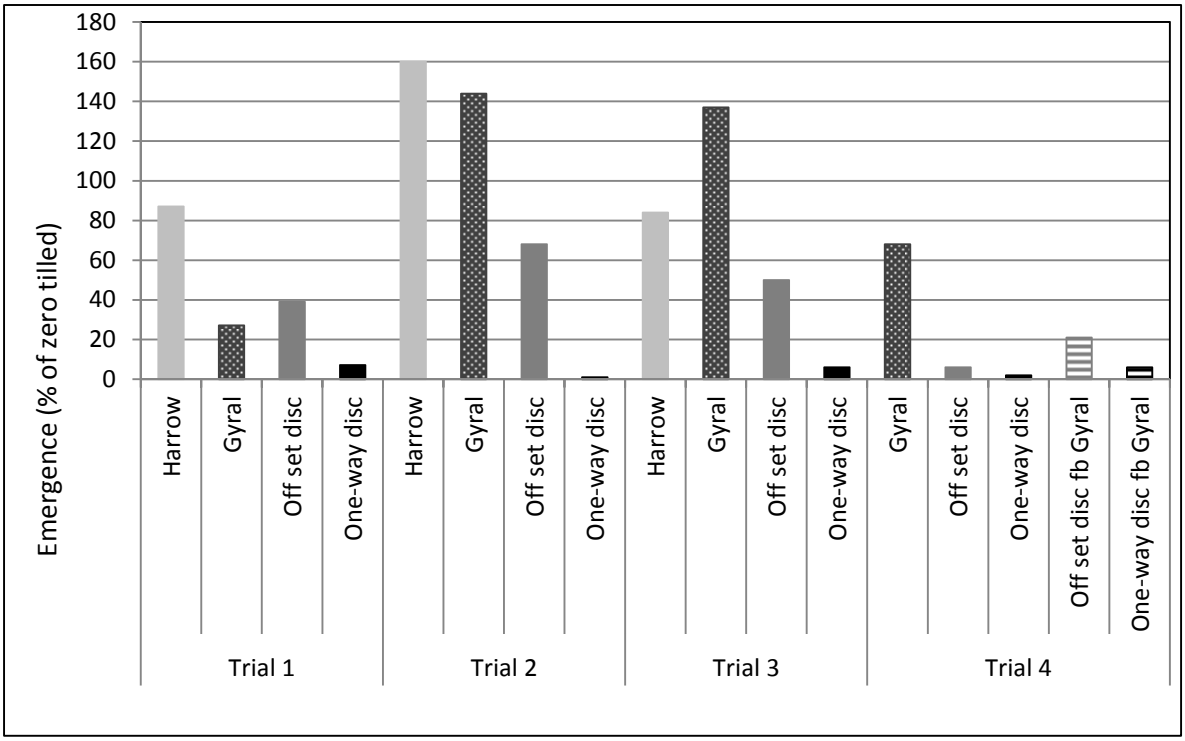

Figure 7. Impact of different forms of tillage on the emergence of common sowthistle as % of emergence in zero tilled plots

Impact of second pass

The second pass with the gyral altered the distribution of glass beads in the soil profile (Figure 5). This in turn resulted in further emergences of all species, but not in all treatments (Figure 8). The emergence of barnyard grass was greatest of all species and while there was an increase in emergence following the gyral double pass after the one-way disc, the same effect was not evident for the gyral double pass after the offset disc. For sowthistle, feathertop Rhodes grass and windmill grass there was a small increase in emergence following the application of the gyral double pass.

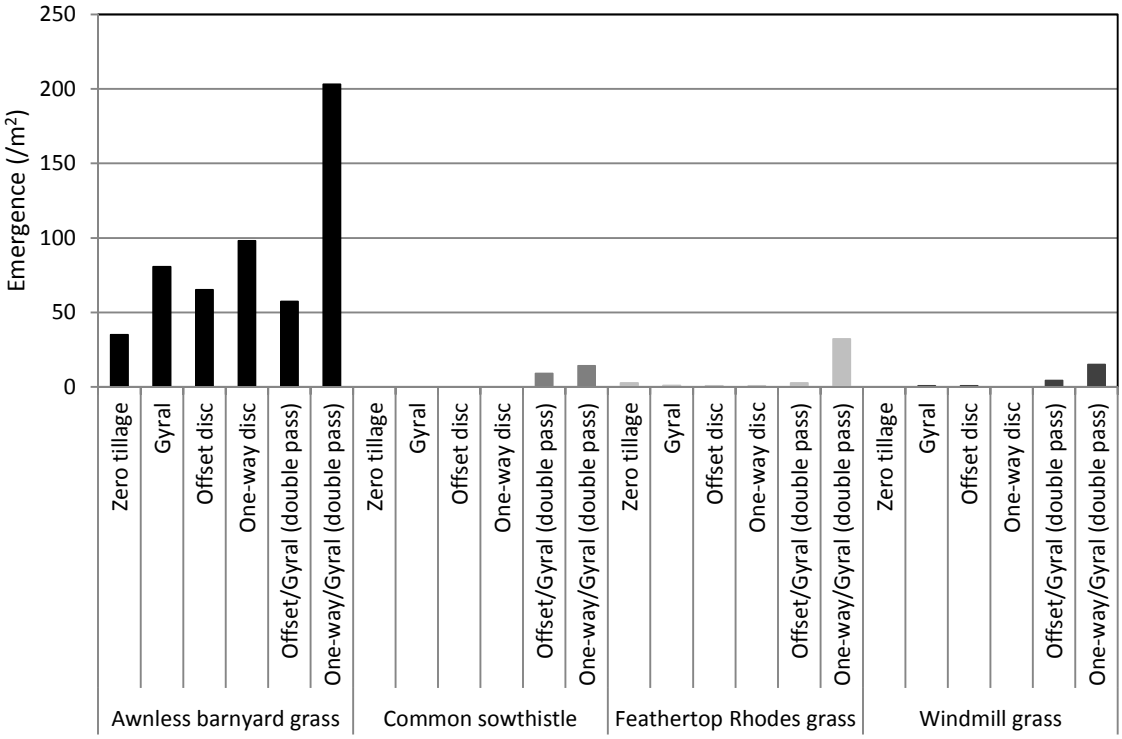

Figure 8. Emergence (/m^2) of key weed northern region weed species after the second pass gyral treatment had been applied following different forms of tillage

New tillage approaches

In a desire to find the balance between retaining the benefits of zero tillage and using tillage to improve weed control, current research is investigating and evaluating the use of robotics and targeted tillage in combination with weed detector technology. Both of these innovative approaches are being investigated to target low density weed populations. Thereby, stopping weed seed set and spread of herbicide resistance, whilst causing minimal soil disturbance.

There are several research groups investigating the role of robotics in the management of weeds. Whilst all are still in the development stages, there has been some very positive progress made. The robots are able to detect weeds, with some systems able to distinguish between weed types (grass vs broad leaf). One research group are developing a system that can apply different weed management tactics, depending upon the weed type present. For example, if it detects a grass weed, it may apply tillage. If the robot detects a broad leaf weed, it may apply herbicide.

Targeted tillage research is developing a hydraulically driven, rapid response tine system for the conduct of spot tillage operations. This approach relies upon a tractor operator but will use technologies already on the market (weed detection, tines).

Management implications

Effective weed management is reliant on an integrated approach. As with any other weed management tactic, the positives and negatives of tillage need to be considered. There have been many positive gains through the adoption of zero till and reduced tillage systems. However, the negative result of herbicide resistance cannot be ignored. We have demonstrated that tillage can have a positive impact in improving weed control. However, tillage across the whole paddock on a regular basis is likely to undo the positive gains achieved through zero tillage. Targeted tillage aimed at disturbing less of the soil and thereby reducing weed seed burial is the optimal approach if tillage is reintroduced in our current farming systems.

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The research undertaken as part of this project is made possible by the significant contributions of growers through both trial cooperation and the support of the GRDC, the author would like to thank them for their continued support.

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Harvest weed seed control systems for the northern region

Michael Walsh, Director Weed Research, University of Sydney

Key words

IHSD, Chaff tramlining, HWSC

GRDC code

UWA00171

Take home message

Chaff tramlining is a simple, effective approach to harvest weed seed control (HWSC) that removes the need for residue burning for HWSC. This system is particularly suited to high residue situations with dedicated tramlines. The newly commercialised iHSD is a more sophisticated approach to HWSC where two hydraulically driven chaff processing mills are neatly fitted to the rear of the harvester. This highly effective system also reduces the need for residue burning but has the added advantage of retaining and redistributing all residues back across the paddock.

Background

There are now several commercially available HWSC methods that effectively target the weed seed bearing chaff fraction during crop harvest. Studies on the efficacy of these practices: narrow windrow burning, chaff carts, bale direct (BDS) and Harrington Seed destructor (HSD) have clearly demonstrated their efficacy in preventing inputs of viable inputs of seed into the seed bank (Walsh et al., 2014; Walsh et al., 2012; Walsh and Newman, 2007; Walsh et al., 2013). However, single system is suited for use across all of Australia's crop production regions and situations. Therefore, there remains a need for the availability of multiple systems and the ongoing development and refinement of current HWSC options. Two relatively new systems, chaff tramlining and the iHSD have recently been introduced and expectations are that their adoption will be high in the northern cropping region.

Chaff tramlining

The practice of concentrating chaff material on dedicated tramlines is termed chaff tramlining where weed seeds are placed in a hostile environment from which it is difficult for germination and emergence. As with all other HWSC systems chaff tramlining focuses on the weed seed bearing chaff fraction and therefore, depending on seed survival has the potential to be similarly effective. In a trial at North Parkes, NSW, comparing narrow windrow burning and chaff lining with conventional harvest both HWSC systems resulted in a 60% reduction in annual ryegrass emergence.

A study evaluating over summer annual ryegrass seed survival at Esperance, WA highlighted the hostile nature of the chaff tramline environment. There was very low seed survival for annual ryegrass seed placed under canola and barley (Table 1). Interestingly though survival was considerably greater for seed placed under wheat chaff. This is despite similar levels of chaff biomass for wheat and barley in particular. At the second time of assessment only annual ryegrass seed beneath the wheat chaff was surviving. At this stage it is not known why there are large differences in seed survival between the different chaff types but obviously this will have a significant effect on the efficacy of chaff tramlining.

Table 1. Survival of annual ryegrass seed at seeding and crop anthesis following placement under a chaff tramline during the previous harvest.

Chaff type	Chaff (t/ha)	Survival (%)	
		At seeding	Anthesis
Canola	31	2	0
Wheat	19	74	10
Barley	18	3	0

Integrated Harrington Seed Destructor (iHSD)

The iHSD is now commercially available as a retrofit system for harvesters in Australia. Testing of the weed seed destruction efficacy of this mill system over the last two seasons has determined that there was similarly high efficacy as the cagemill used in the trail behind HSD system (Table 2). Very high level s of seed destruction were recorded for the four dominant species of Australian cropping, annual ryegrass, wild radish, wild oats and brome grass.

Table 2. Effect of iHSD mill processing of wheat chaff on the seed mortality of four weed species.

Weed species	Seed kill (%)	SE
Annual ryegrass	93	0.7
Wild radish	99	0.2
Wild oats	99	0.1
Brome grass	99	0.2

High weed seed destruction levels were recorded when the iHSD mill system was evaluated under commercial harvest conditions. During the harvest of canola and barley crops known numbers of annual ryegrass, wild radish, wild oat and brome grass seed were introduced to an iHSD mill fitted to a class 9 harvester. Processed chaff was collected and sieved and sorted to recover viable weed seeds. In all instances there was 99% kill of the introduced weed seed.

Summary

The number of HWSC options continues to grow as producers look to utilise this approach to weed control in cropping systems. The iHSD is now commercially available following several years of field stationary mill testing that have proven its efficacy and commercial capacity. Chaff tramlining is becoming widely adopted as growers move to tramline systems and look to reduce residue burning in their production systems. Both approaches are highly effective weed control tools and can provide considerable support for herbicide based weed management programs.

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Stopping spray drift – the Australian Ground Spray Calculator is a new decision-support tool to help growers better manage their spraying operations

Andrew Hewitt, Chris O'Donnell, Gary Dorr, Jason Ferguson and Rodolfo Chechetto, University of Queensland

Key words

Spray drift, spray calculator, nozzle, DRT, buffer

GRDC code

UQ00060, UQ00072

Take home message

The Australian Ground Spray Calculator is a decision-support tool that provides spray applicators with information on droplet size, target coverage and drift potential specific to particular nozzles and tank mixes. Spray applicators now have a unified tool that provides science-based reliable information to help with their drift management strategies.

Introduction to spray drift

Just because a spray is classified as "Coarse", it can still include up to 10 or more out of every 100 L in "fines". In other words, for an application rate of 100 L/ha, 10 of those 100 L could potentially be available to drift off-target in unfavourable conditions. In a relatively high wind speed in an unstable atmosphere, this driftable part of the spray could move relatively large distances, but is likely to disperse more than under conditions of little or no wind in a local surface temperature inversion where the total drift loss and distance may be similar to that of the high wind scenario but the concentrations of droplets on the ground at any one distance can be hard to predict. In some places there may be no droplets, while in other places there could be relatively high concentrations of this deposition drift under stable air conditions. The above hypothetical cases of unstable and stable air may have involved the same amount of airborne drift but very different patterns of deposition drift.

Managing drift

Pesticide exposure risk assessors usually assume that the air is unstable because applications under conditions of local surface temperature inversions are forbidden on labels and in most pesticide application regulations. When stable air scenarios are ignored in risk assessment, the normal trend for exposure is that airborne and deposition drift increase with higher wind speeds. However, the label tends to focus on Spray Quality for alerting the applicator how to avoid drift with a particular product. For example, phenoxy herbicides are usually labelled for requiring a spray that is at least Coarse in Spray Quality.

Improvements to regulations

However, through the work of UQ and others, regulations could soon become more flexible to allow an applicator to manage drift in other ways than just droplet size, and even to reduce the size of any required no-spray buffer zones by using a suitable Drift Reduction Technology, or DRT instead or, or as well as the standard nozzle options. DRTs may include novel designs of hardware options (e.g. better nozzles, atomisers or shields, shrouds, air-assistance, etc.), or formulation chemistries (e.g. some emulsion and other chemistries). In some cases, landscape features such as hedges or netting

may be allowed as DRT shields or barriers to intercept drift and protect sensitive areas downwind and beyond their locations.

Spray Performance Calculator

One aim of the Spray Performance Calculator is to be ready to show DRTs once the Australian Pesticide and Veterinary Medicines Authority (APVMA) announces and launches its DRT scheme for Australia (this is expected late in 2016, at which time the Spray Performance Calculator will be updated accordingly). The Calculator already includes an output called "Drift Potential". This indicates the relative drift potential of the spray. In the example below, the spray selected by the end-user was classified as Coarse for Spray Quality, but had a relatively low drift profile and exposure level. Note that the percent "fines" in this case is almost 80% lower than the default ~13% for the reference category of "Coarse" sprays. This is important because if an applicator can select a Spray Quality that is finer than the Drift Potential then he can often increase the potential spray coverage at targets without needing to increase his application volume rate.

Factors influencing performance of a pesticide spray

It has been known for many years that the choice of nozzle, spray pressure and tank mix composition including active ingredient and adjuvant products can have a large effect on the performance of a pesticide spray for efficacy and targeting. In general, the coarser the spray, the lower its drift potential but the lower the potential coverage on target surfaces, plants and pests. This is where factors like increasing the water volume rate can help boost droplet numbers, but how does an applicator know how to get the balance between the spray coverage and avoidance of drift when faced with so many variables?

In particular, adjuvant effects are often non-intuitive with some adjuvants increasing the fine droplets in a spray and others reducing these "fines". To make things more complicated, one trend may be seen with one nozzle but a completely different trend with a different nozzle. To help take the guesswork out of spray performance and also help comply with new spray drift management requirements, researchers at The University of Queensland (UQ) have measured the performance of over a thousand combinations of nozzle x pressure x tank mix that cover most Australian grain crop spraying scenarios and assembled these data into a new Australian Spray Performance Calculator© that can help show the key performances of a spray in relative terms: a) coverage in number of droplets/cm^2, b) coverage after droplets wet a leaf, c) Spray Quality in terms of droplet size classification and d) Drift Potential. This calculator differs from previous spray performance tools in Australia and the USA which tended to only state the Spray Quality.

Additional information in the Calculator shows the droplet size spectrum and where it sits relative to the American Society for Agricultural and Biological Engineers (ASABE, formerly ASAE) S572 reference spray boundary curves. ASABE S572 "Spray Nozzle Classification by Droplet Spectra" is a system that allows sprays and nozzles to be classified by Spray Quality into categories of relative size. In the example below, the volumetric droplet size spectrum curve (solid blue line) crosses between different size classes depending on the volume fraction/ droplet size region. The ASABE S572 scheme requires the classification be assigned to the finest category in this distribution which resulted in a Coarse spray designation.

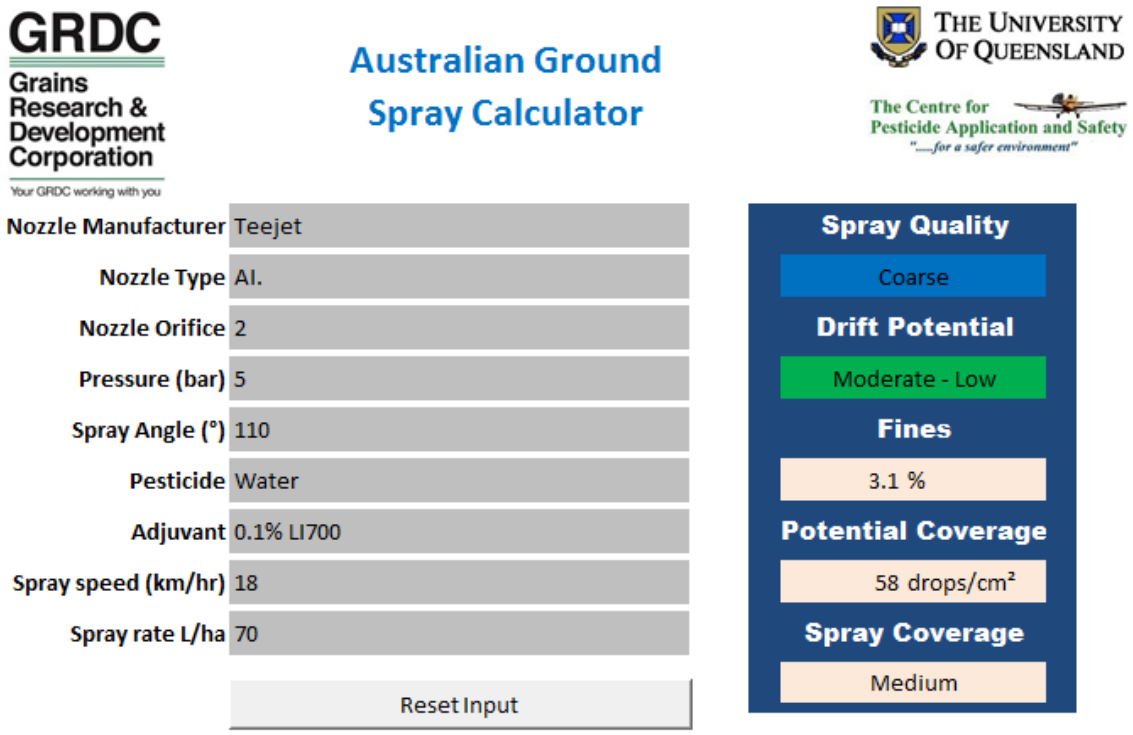

Figure 1. Spray performance calculator

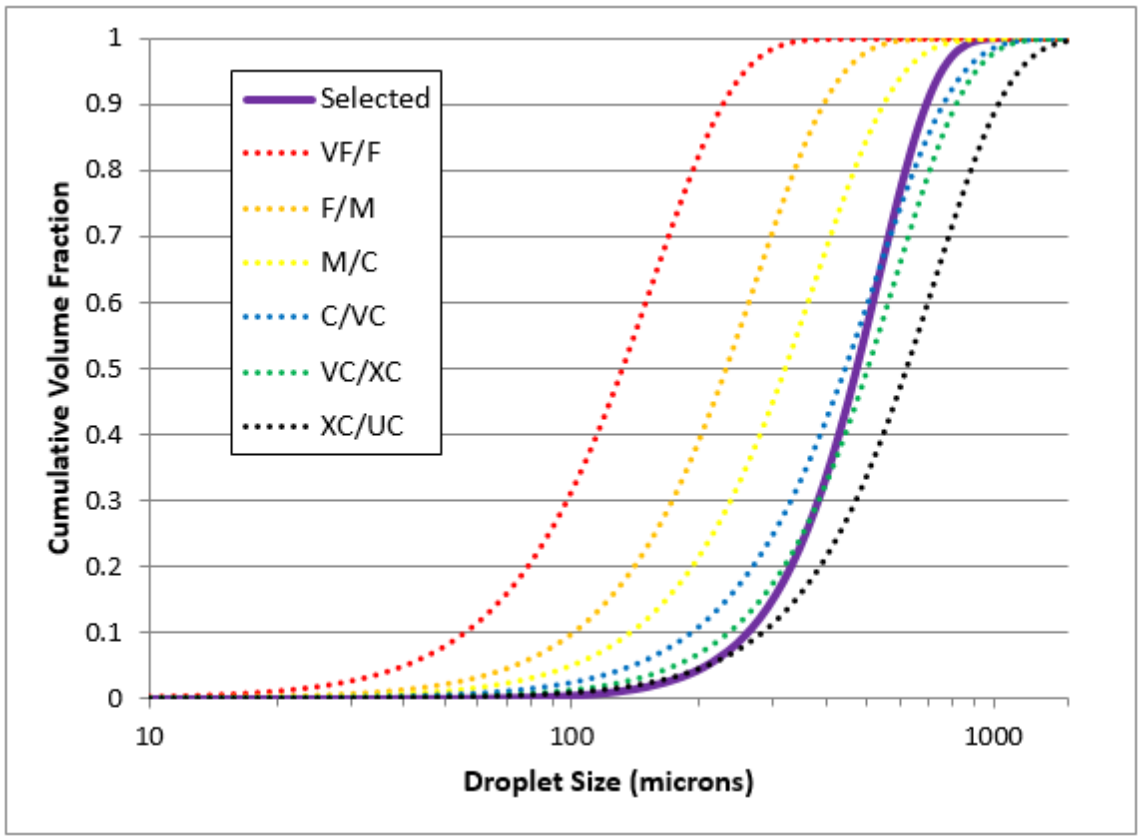

Figure 2. Droplet size spectrum for the example scenario from the calculator (solid line) compared to ASABE S572 reference curves which are (from L to R) VF/F, F/M, M/C, C/VC, VC/XC, XC/UC.

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