DALBY, QLD &
NARRABRI, NSW

TUESDAY 2 and THURSDAY 4 MARCH 2021

GRAINS RESEARCH UPDATE

DRIVING PROFIT THROUGH RESEARCH

GRDC
GRAINS RESEARCH & DEVELOPMENT CORPORATION

GRDC 2021 Grains Research Update Welcome

If you are reading this, then chances are you are sitting in one of the GRDC's first face-to-face events since COVID-19 changed our lives.

Welcome.

We at the GRDC understand how challenging the past year has been for all Australians, but we also appreciate how well positioned agriculture has been to respond to and work through the restrictions that have come with this global pandemic.

Across many areas of Queensland and New South Wales, an improvement in seasonal conditions has also provided a much-needed reprieve for growers, advisers, agronomists, researchers and those associated with the grains industry.

With that positive change in circumstances comes a thirst for the latest information and advice from grains research and development – we trust that these GRDC Grains Research Updates will help guide your on-farm decisions this season and into the future.

While COVID-19 has forced temporary changes to our traditional Update locations and audience numbers, these events still offer the high quality, seasonally relevant research, development and extension information you have come to depend on. This year our Updates will also be live streamed to ensure the information is available to all who need it.

We would like to take this opportunity to thank our many research partners, who, like growers and advisers, have gone over and above to continue to work in situations constricted by COVID-19 regulations.

Challenging times reinforce the importance of rigorous, innovative research that delivers genuine gains on-farm. For more than a quarter of a century the GRDC has been driving grains research capability and capacity with the understanding that high quality, effective RD&E is vital to the continued viability of the industry.

Sharing the results from this research is a key role of the annual Updates, which are the premier event on the northern grains industry calendar and bring together some of Australia's leading grains research scientists and expert consultants.

To ensure this research answers the most pressing profitability and productivity questions from the paddock, it is critical the GRDC is engaged with and listening to growers, agronomists and advisers.

For the past five years we have been doing that from regional offices across the country. Through the northern region offices in Wagga Wagga and Toowoomba and a team of staff committed to connecting with industry, we are now more closely linked to industry than ever.

This year we have less people on the ground – as a result of COVID-19 restrictions – but more than ever we are available to listen and engage with you. So if you have concerns, questions or feedback please contact our team directly (details on the back of these proceedings) or email northern@grdc.com.au.

Regards,
Gillian Meppem
Senior Regional Manager – North

GRDC
GRAINS RESEARCH & DEVELOPMENT CORPORATION

Dalby – Tuesday 2 March

Start: 9am, finish 4:20pm. (Please note times are Queensland time)

Time	Topic	Speaker(s)
9:00 AM	Welcome	GRDC
9:20 AM	**Cereal pathology update** - learnings from 2020; planning for 2021; latest trial outcomes; barley Powdery mildew in 2020; is stripe rust the death knock for some varieties?	Lislé Snyman (DAF Qld)
9:50 AM	**Satellites and other useful data sources that are ready for immediate benefit for on-farm decision making.**	Tim Neale (Data Farming)
10:20 AM	Morning tea	
10:50 AM	**ConstraintID** – a free web-based tool to identify areas where soil constraints are most likely to be limiting crop yields.	Yash Dang (UQ)
11:20 AM	**Amelioration for sodicity** - deep ripping and soil amendment addition across NSW and Qld. Engineering challenges. Yield responses to ripping, gypsum and organic matter placement in constrained soils.	Dave Lester (DAF Qld)
11:50 AM	**Identification of crop-specific and varietal tolerance limits to sodicity.**	Jack Christopher (UQ)
12:15 PM	Lunch	
12:55 PM	**Optimising chickpea sowing and flowering dates for maximum yield.**	Allan Peake (CSIRO)
1:25 PM	**The drivers of phenology for more profitable sowing decisions** - balancing risk of abiotic stresses when selecting cereal varieties to optimise yield potential in southern Qld.	Felicity Harris (NSW DPI)
2:00 PM	**Fall armyworm** - an Update.	Melina Miles (DAF Qld)
2:25 PM	Afternoon tea	
2:55 PM	**Herbicide resistance survey results.** Fleabane, feathertop Rhodes grass, barnyard grass, sowthistle, windmill grass.	Michael Widderick (DAF Qld)
3:20 PM	**Glyphosate** resistance and optimising performance; optimising **clethodim** efficacy; update of **wild oat resistance** tests from NSW/Qld.	Peter Boutsalis (Plant Science Consulting)
4:00 PM	**Weed ecology, seed persistence and retention** - implications for management and harvest weed seed capture. (Barnyard grass, feathertop Rhodes grass, windmill grass and sow thistle). **Also: New data on resistance in feathertop Rhodes grass and sow thistle.**	Bhagirath Chauhan (UQ/QAAFI)
4:30 PM	Close	

(Agenda subject to change)

Narrabri – Thursday 4 March, 2021

Start: 9am, finish 4:50pm. (Please note times are NSW summer time)

Time	Topic	Speaker(s)
9:00 AM	Welcome GRDC	GRDC
9:10 AM	**Wheat 2030** opportunities, immediate issues with feed market recovery after the drought and feed grain demand projections. - 2030 production and trade projections and implications - 2030 managing risk and trade diversification	Ross Kingwell (AEGIC)
9:50 AM	**Getting phenology right, what's next?** Project learnings from 2017-20, comparing wheat and barley responses and tactical agronomy to increase yield.	Rick Graham (NSW DPI)
10:35 AM	Morning tea	
Weeds		
11:05 AM	**Are wild oats adapting to environmental stresses and conservation agriculture?**	Mohammad Ali (PhD - UQ)
11:30 AM	**Herbicide resistance:** o Optimising glyphosate performance o What's new in resistance in NNSW? o Optimising clethodim efficacy o Changing germination patterns of annual ryegrass o Stripper fronts and weed seed collection	Peter Boutsalis (Plant Science Consulting) & John Broster (CSU)
12:25 PM	Lunch	
Agronomic strategies to optimise pulse productivity in 2021		
1:05 PM	**Optimising chickpea sowing and flowering dates for maximum yield.**	Allan Peake (CSIRO)
1:30 PM	**Why were pulse viral diseases so bad in 2020?** Which viruses, what vectors and with what crop impact? Management strategy for 2021	Joop Van Leur and Zorica Duric (NSW DPI)
2:00 PM	**Impact of Ascochyta on chickpea when disease occurs at different growth stages**	Kevin Moore (NSW DPI)
2:25 PM	**A new desi variety 'CBA Captain⁽ᴰ⁾' for NNSW and SQld in 2021**	Kristy Hobson (NSW DPI)
2:50 PM	Afternoon tea	
3:20 PM	**How wide is the distribution of Russian wheat aphid (RWA) in northern NSW** and is sorghum an alternative summer host?	Zorica Duric (NSW DPI)
3:45 PM	**Heat tolerance in cereals** - best performing lines, new material, mechanisms of tolerance and in season recommendations	Rebecca Thistlethwaite (University of Sydney)
4:15 PM	**Cereal disease bingo!** Learnings from 2020 and planning for 2021.	Steve Simpfendorfer (NSW DPI)
4:50 PM	Close	

(Agenda subject to change)

Contents – DALBY – TUESDAY 2 MARCH 2021

Contents – NARRABRI – THURSDAY 4 MARCH 2021

Compiled by Independent Consultants Australia Network (ICAN) Pty Ltd.
PO Box 718, Hornsby NSW 1630
Ph: (02) 9482 4930, E-mail: northernupdates@icanrural.com.au

Cereal pathology update – learnings from 2020, planning for 2021

Lislé Snyman, Department Agriculture and Fisheries Queensland

Key words

barley, powdery mildew, virulence, pathogen, loose smut, covered smut, seed-treatment, fungicide

GRDC codes

National Variety Disease Screening (NVT), DAQ2005-004, DAQ1907-001

Take home messages

- High levels of powdery mildew infection were observed in 2020 barley crops
- Powdery mildew is a highly variable pathogen of barley with virulence varying in response to varietal resistance
- Continuous barley cropping increases the risk of stubble-borne diseases
- Management strategies for foliar diseases include resistant varieties, crop rotation, seed treatment, regular crop monitoring and timely fungicide application
- Resistance to fungicides have been reported in powdery mildew, net form of net blotch (NFNB) and spot form of net blotch (SFNB) in Australia
- Limit fungicide application by spraying only when necessary, rotate fungicides with different modes of action and use recommended rates
- Seed-treatment will provide protection against seed-borne diseases, smut and early development of powdery mildew
- Effective seed treatment depends on product choice, application rate and efficacy of application.

Background – learnings from 2020

Rainfall in the 2020 season in Queensland was lower than expected, hence disease incidence was also lower than anticipated.

However, conditions resulted in high levels of powdery mildew (PM) infection, particularly on varieties with low levels of resistance. Powdery mildew (*Blumeria graminis* f. sp. *hordei*) is a disease synonymous with barley cultivation world-wide. Under mild humid conditions, it will infect leaves and leaf sheaths of plants. It is easily recognisable by the white, fluffy mycelial growth, particularly on upper leaf surfaces. Older colonies turn dull grey and often produce small, black fruiting bodies. Environmental conditions in Qld generally become unfavourable for the pathogen and disease does not persist to adult plant stages. Consequently, yield losses are usually below 15%. In the 2020 season, infections persisted later into the season, despite conditions becoming less favourable.

Both loose smut and covered smut were detected in a number of barley crops in Qld in 2020, with losses in yield and quality reported. Affected crops had not been treated with an effective seed treatment. Seed-treatment applied effectively would have avoided yield loss.

Net blotch was present at low levels and was most conspicuous in irrigated crops. Net blotch in barley is caused by one of two forms of *Pyrenophora teres* (*P. teres*). Net form net blotch (NFNB) is caused by *P. teres* f. *teres* (*Ptt*) and spot form net blotch (SFNB) is caused by *P. teres* f. *maculata* (*Ptm*). The two forms share the same shape and structure (morphology) and can only be distinguished by plant symptoms and molecular characterisation. Symptoms of NFNB are characterised by net-like dark brown necrotic lesions, whereas SFNB symptoms are characterised by dark circular or elliptic brown spots surrounded by a yellow chlorotic area.

Net form net blotch occurs regularly in the northern region and samples are collected from crops annually. However, only 5 samples of NFNB were collected or submitted for pathotyping in 2020. SFNB was present at very low levels in isolated crops, unlike 2019 where despite dry conditions SFNB was widespread in Qld barley crops. Moisture stress may have contributed to crops being more vulnerable to infection.

Spot blotch (SB) (*Bipolaris sorokiniana)* has previously been confined to the warmer and more humid coastal areas of Qld. In recent years however, an increased number of SB samples have been collected from Central Qld (CQ). This could be attributed to the increased area sown to barley in CQ. With an increasing area sown to barley, the incidence of SB is expected to increase under favourable conditions and may become a significant yield constraint.

Powdery mildew

Powdery mildew (PM) survives between seasons on volunteer barley and barley stubble. When conditions become less favourable, the pathogen undergoes sexual reproduction and forms fruiting bodies (cleistothecia) in existing colonies. These persist until the new growing season and release ascospores under favourable temperature and moisture conditions, infecting the new crop. The mildew colonies produce airborne conidia (asexual spores) that spread the disease within and between crops.

A survey of Australia's barley powdery mildew population was conducted by Dreiseitl and Platz in 2010 and 2011. This study identified 27 pathotypes and 16 major genes effective against all PM isolates tested. Largely as a result of its sexual reproductive stage, the pathogen can evolve rapidly, resulting in new pathotypes which can damage previously resistant varieties. Since then, pathotypes that are virulent against the resistance genes *Mla3*, *Mla9*, *Mla12* and *MlLa* have been identified, thus reducing the number of effective major resistance genes.

Virulence for *MlLa* is responsible for the increased disease levels occurring in certain varieties i.e. Commanderᶲ, Mackay, Compassᶲ, Hindmarshᶲ, and La Trobeᶲ. This gene is present in many of the current varieties, with high levels of PM observed on Rosalindᶲ and Spartacus CLᶲ in Qld in 2020.

The *mlo* gene provides resistance to all isolates. Varieties that have this gene include Grangerᶲ, Westminsterᶲ and RGT Planetᶲ. Other commercial varieties resistant to powdery mildew and their effective resistance genes include Fairviewᶲ (*Mla13*), Flindersᶲ (*Mla1*) and Scopeᶲ (*Mla7*).

Smut

Barley hosts two species of smut – loose smut (*Ustilago nuda)* and covered smut (*Ustilago hordei*). In both, infection results in florets producing thousands of spores, instead of grain. Spore masses are encased in a membrane. This membrane is quite fragile in loose smut and ruptures soon after head emergence, releasing the spores. However, in covered smut, the membrane is much more persistent, often breaking during harvesting.

Both species were detected in a number of barley crops in Qld in 2020. Loose smut has been reported in previous years, with crops of the Hindmarshᶲ lineage e.g. Hindmarshᶲ, La Trobeᶲ and Rosalindᶲ, often infected.

Loose smut is most conspicuous around flowering when infected heads, bearing a mass of dark brown to black sooty spores, are visible. In plants infected with loose smut, the membrane ruptures soon after heading, releasing airborne spores which infect surrounding florets. Infection occurs under moist conditions at temperatures around 16 – 22°C. Florets are susceptible to infection from flowering to about one week after pollination. Germinating spores infect the ovary and the fungus survives as mycelium within the embryo of the infected seed. Once infected seed is sown, it germinates and carries the fungus in the growing point of the plant becoming visible as a black spore mass at head emergence. Loose smut is well adapted for survival with infected plants usually being slightly earlier than healthy plants, ensuring an adequate supply of inoculum when the bulk of the crop is flowering.

Heads infected with covered smut frequently emerge later than healthy heads and often being shorter and harder to see. As with loose smut, grains are replaced with a mass of black powdery spores. These are released during the harvesting process and contaminate healthy grain. The spores germinate after planting, infecting emerging seedlings, growing through the plants where they eventually replace the grain with spores. The fungus is favoured by temperatures of 14 – 25°C.

Loose smut is exclusively internally seed-borne, while covered smut is either externally seed-borne or survives in the soil. The life cycle of loose smut in barley is the same as in wheat; however barley loose smut will not infect wheat and vice versa.

Disease management

Barley foliar pathogens are a significant challenge to the grains industry and a major constraint to profitable barley production, affecting both yield and quality. Many of these pathogens are genetically and pathogenically diverse, able to reproduce sexually and can rapidly develop new virulence and overcome genetic resistance.

The adoption of stubble retention practices has led to an increase in the incidence of stubble-borne diseases. Planting successive barley crops in the same paddock increases pathogen incidence.

Growing a high yielding, well adapted, resistant variety provides the most economic and environmentally friendly means of disease control. Genetic resistances need to be durable to provide long-term protection. The PM resistance gene *mlo* has been effective in Europe for more than 50 years but has not been widely adopted in Australian breeding programs due to its association with lower yields in the Australian environment. It is however present in some varieties, mostly from European descent including Granger⦶, Westminster⦶ and RGT Planet⦶. Other commercial varieties resistant to powdery mildew and their effective resistance genes include Fairview⦶ (*Mla13*), Flinders⦶ (*Mla1*) and Scope⦶ (*Mla7*).

In susceptible varieties where yield potential is high, fungicidal control of PM can be justified. Foliar fungicides should be aimed at protecting the top two leaf layers. The identification of PM populations that are resistant to or have reduced sensitivity to Group 3 (DMI) fungicides in WA, highlights the importance of resistance breeding.

Net form net blotch is best controlled by sowing varieties rated MS or better and a combination of cultural practices. The NFNB pathogen persists on plant residue. Cultivation of the same variety will lead to an increase in the presence of pathotypes virulent on that particular variety and put increased pressure on effective resistance genes. Best practice includes crop rotation with non-host crops such as wheat, canola and chickpea. NFNB is also seed-borne and can spread with infected seed. Various seed treatment products are registered for NFNB control.

Resistance to Group 7 and Group 3 fungicides has been identified in NFNB populations in SA and WA, respectively, with reduced sensitivity identified in WA populations to other Group 3 (DMI) fungicides. To ensure that fungicides remain effective, it is important to limit fungicide application by spraying only when necessary, rotate fungicides with different modes of action and use fungicides at recommended rates. Avoid using tebuconazole as a stand-alone product in barley to avoid indirect fungicide resistance selection. By applying it for PM control, you can indirectly select for NFNB or SFNB isolates resistant to tebuconazole, without the intention of controlling those diseases. Isolates resistant to fungicides can be spread through infected seed. It is beneficial to all to ensure that we use fungicides in such a way that we protect their longevity.

Fungicide applications are more effective if applied before disease becomes established in the crop. This requires regular monitoring to ensure crops can be sprayed at the first sign of disease. When conditions are favourable for disease development, more frequent crop inspections will be needed and repeat fungicide applications may be necessary.

The level of smut in a crop is a function of

- Varietal susceptibility
- The number of grains infected in the previous seed crop
- The efficacy and rate of the seed treatment applied and
- The precision of the seed treatment process.

Resistance to smut is available; but has never been a priority of Australian barley breeding programs. Seed treatment has provided economical control in the past and is likely to continue to do so. Several fungicides are registered for the control of smut; but the levels of control vary among products.

Conclusion and 2021 planning

The absence of many diseases in 2020 in the northern region does not mean that we can be complacent. With favourable environmental conditions, pathogens will continue to cause yield and quality loss and we have to make the right decisions to ensure that we can stay ahead of disease development and the evolution of the pathogen.

Continuous monitoring of the pathogen populations provides information on the virulence in the Australian pathogen populations and aids in the identification of effective resistance for use in the development of resistant varieties. There are still a range of major resistance genes effective against our powdery mildew population and increasing evidence of useful adult plant resistance genes. These need to be used in a manner that ensures the resistances will be durable. We know that *mlo* resistance is effective and durable and encourage Australian breeders and pre-breeders to utilise it as a source of resistance.

The reappearance of smut in barley is a reminder to maintain effective fungicide treatment of planting seed. If seed is sourced from a crop known to have been infected with loose smut, it would be useful to treat seed at the higher recommended rate. The increased incidence may be due to a variety of reasons, including good infection conditions, application of lower rates of fungicide or ineffective application or varietal susceptibility. Quality routine seed treatment should continue to provide effective smut control.

References

2020 Queensland winter crop sowing guide

https://grdc.com.au/queensland-winter-crop-sowing-guide

2021 Queensland winter crop sowing guide

https://grdc.com.au/2021-queensland-winter-crop-sowing-guide

Dreiseitl A and Platz G (2012) Powdery mildew resistance genes in barley varieties grown in Australia. Crop and Pasture Science 63, 997 – 1006.

Acknowledgements

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.

Contact details

Lislé Snyman
DAF QLD
Hermitage Research Facility, 604 Yangan Rd, Warwick, Qld
Ph: 07 4542 6761 / 0428324932
Email: lisle.snyman@daf.qld.gov.au

⦶ Varieties displaying this symbol beside them are protected under the Plant Breeders Rights Act 1994.

Satellites and other useful tools that are ready for immediate benefit for on-farm decision making

Tim Neale, DataFarming

Key words

satellite imagery, precision agriculture, sensors, adoption

Take home message

Growers and agronomists have access to free (and paid) precision agriculture (PA) tools to obtain immediate benefits for their crop management. Satellite imagery represents one of the simplest entry points to get started on the journey of precision agriculture. Many Australian farms have yield variation of more than 300% in every single paddock. Combined with targeted soil and plant sampling, a deeper understanding of the factors impacting on production can be identified and remediated. Coupled with that, variable rate technology (VRT) offers the ability to better target crop inputs.

Barriers to adoption

In a GRDC/CSIRO survey several years ago, it showed that whilst around 75-80% of farmers had adopted auto-steer GPS systems on machinery, only 4% of farms had viewed a satellite image of their property, and only 10% were using variable rate technology (Llewellyn and Ouzman, 2014). DataFarming set about to identify the barriers to adoption of digital agriculture technologies, and narrowed them down into five main areas:

1. Cost of entry
2. Complexity of the systems available
3. Ease of use and user experience
4. Connectivity (internet, and between equipment brands) and
5. Not getting instant value.

Whilst many farmers embrace digital ag technologies, agronomists are making more and more complex decisions on increasingly bigger farms and paddocks; so there is a great opportunity to become far more efficient in the way we check, manage and treat crops.

Getting started

The first step is to get satellite imagery of your paddocks whilst the crop is growing. With the availability of 10 metre resolution data every five days anywhere in the world, essentially for free, this is a great starting point. The key difference is that satellite imagery shows impacts beyond what our eyes can see – so we can detect crop problems earlier, and easily measure the impacted area. A crop growth index called NDVI which measures in the infrared spectrum, picks up difference in crop health/greenness/biomass. This helps direct where to look when scouting or checking crops. See below (Figure 1) for an example from the DataFarming platform – where the blue areas (centre) of the paddock are high growth, and the areas in red (left and right) are poorer growth.

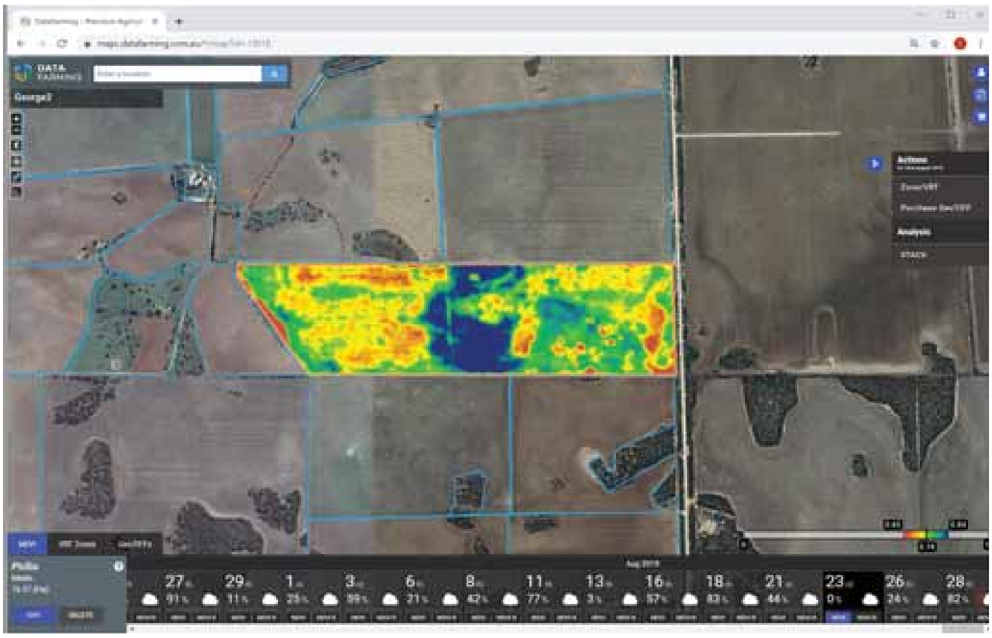


Figure 1. An example from the DataFarming platform – where the blue areas (centre) of the paddock are high growth, and the areas in red (right and left) are poorer growth.

After three years (up to the end of 2020) there are now 20,000 farms loaded in the DataFarming platform covering almost ¼ of the Australian grains industry, and 20M acres of paddock level data processed.

Targeted soil and plant sampling

Whilst technologies such as EM (electromagnetic) mapping have been around for decades, only a few key contractors offer the service in Australia. With the advent of new tools, any agronomist can now capture soil type data using EM from the comfort of the ute. EM typically measures down to 1.5m into the soil to detect differences in clay content, moisture content, and soil salts. Below (Figure 2) is an example of one of these tools, and the resultant data on the right.

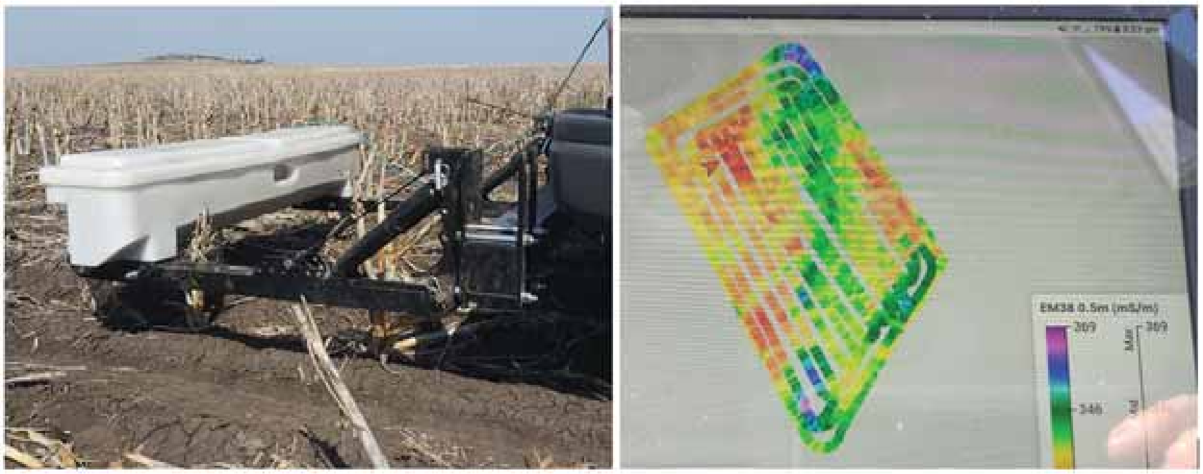


Figure 2. An example of an EM machine on the back of a ute (left) and resulting data map (right)

For agronomists collecting soil samples, the APAL Farm2Lab app takes the out the paperwork and streamlines the whole process. It doesn't make the practical part of sampling any easier, but certainly speeds up the process and improves efficiency. You can add zonal layers at the planning stage, before entering the paddock, which helps select the right spot to sample. Once sites are selected and you determine the suite of tests you want, the points are available in an offline app which directs you in the field. Once collected the geo-referenced results are presented back in the app for easy viewing and understanding. The results can also be pushed to BackPaddock or Agworld software for interpretation and building recommendations.

The image (Figure 3) below shows zone maps in the Farm2Lab app, ready for soil sampling.

Figure 3. Zone maps in the Farm2Lab app, ready for soil sampling

Results can also be displayed in other platforms. The image below (Figure 4), shows the resultant soil test levels overlaying the NDVI imagery in the Summit fertiliser app. This enables farmers and agronomists to get better value out of soil testing, as well as understand and alleviate production limiting factors.

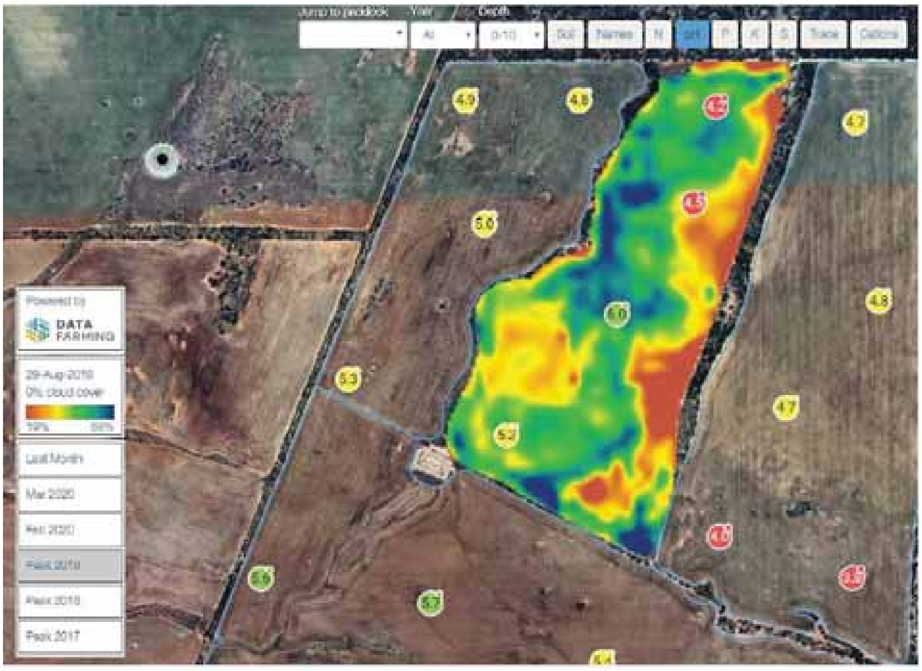


Figure 4. Soil test levels overlaying the NDVI imagery in the Summit fertiliser app

References

Llewellyn, R. and Ousman, J. (2014) Adoption of precision agriculture-related practices: status, opportunities and the role of farm advisers. Accessed from: https://grdc.com.au/__data/assets/pdf_file/0024/208653/adoption-of-precision-agricultural-related-practices-status-opportunities-and-the-role-of-farm-advisers-2014.pdf.pdf

Acknowledgements

DataFarming acknowledges the funding it has received through the GrainInnovate fund managed through Artesian Venture Partners.

Contact details

Tim Neale
DataFarming
P.O. Box 253, Highfields 4352
Ph: 0409 634 006
Email: tim@datafarming.com.au

ConstraintID – a free web-based tool to identify areas where soil constraints are most likely to be limiting crop yields

Yash Dang[1], Tom Orton[1], David McClymont[2] and Neal Menzies[1]

[1] School of Agriculture and Food Sciences, The University of Queensland
[2] DHM Environmental Software Engineering Pty Ltd

Key words

ConstraintID, soil constraints, crop yield, decision support tool, remote sensing, Landsat

GRDC code

UOQ1803-003RTX

Take home messages

- Better knowledge and understanding of the variation of past crop yields (both spatially across a paddock, and temporally from season to season) can help growers make better management decisions to improve yields in future
- Remote-sensing data compiled from earth-observing satellites provide a valuable information source about past crop yields, allowing us to look through multiple years of imagery for consistent patterns through time – that is, spatial patterns of yield variation that repeat season after season
- Such consistent patterns might imply the presence of some kind of soil constraint limiting yields in parts of the field
- This web-based tool enables growers to easily look at processed remote-sensing data representing past crop yields in their paddocks. The tool will show if there are any consistent spatial patterns in the data and will compile and present maps of soil constraints for comparison. The data on these driving factors could help the user to interpret the variation shown by the remote-sensing data.

Introduction

Within-field spatial variation of crop yields can occur for various reasons, but perhaps the most important driver of this variation is the soil. There are different management strategies available to growers to address soil constraints (amelioration, adjusting inputs to match potential, switching to growing tolerant species), but a necessary first step is the identification of affected areas (Dang et al., 2010).

Persistent spatial patterns of yield variation within fields that have otherwise been managed uniformly (spatially) provide an indication that soil constraints are driving variation. Remotely sensed data from satellites provide a valuable information source for helping with the task of identifying persistent yield patterns (Orton et al., 2018). In particular, the Landsat series of satellites provide a very useful dataset for several reasons:

(i) The long history (in this area of data collection from 1999-present), which allows persistent spatial patterns of within-field yield variation (similar patterns repeating season after season) to be identified
(ii) The time between repeated images of the same area (8 days when two of the Landsat satellites are operating), which allows growth patterns within a season to be identified
(iii) The spatial coverage of satellite data (data readily available across the whole region)

(iv) The spatial detail (30-m pixel sizes), which is detailed enough to allow within-field patterns of spatial variation to be identified, and
(v) It is freely available.

While Landsat imagery provides an excellent source of raw data, there are a number of processing and analysis steps required to extract the potentially useful information. We have developed a freely available online software tool, ConstraintID (http://constraintid.com.au), to make this task easy for growers, and to present results in a way that is straightforward to interpret. The tool should assist growers with identification of potentially constrained areas, giving them the information to make more informed management decisions. Here we present an illustration of the software tool applied to a field in northern NSW.

Illustration

Remote-sensing data to give crop performance information for all years

The software undertakes the spatial analysis within the confined boundary of a field. It is important that there has been consistent management across the field, so that spatial differences due to management will not be confused with differences due to soil constraints. The software then collates all of the remote-sensing data for this field, and for each year from 1999 to the latest complete season, generates a map of an index representing crop performance. This aim of this index is to show, for any given year, which parts of the field were above average and which parts were below average. In some of these years a winter crop might not have been grown, so the software looks at the growth patterns within each season to check whether the field has been cropped and determines if it that years observations should be included in the analysis. For illustration, Figure 1 shows the resulting maps for the years 2005-2010 at Sunbury. In this case, the software deemed that there was good evidence of 2005, 2006, 2008 and 2009 being cropped years (shown by the green ticks), so these would be included in further analysis.

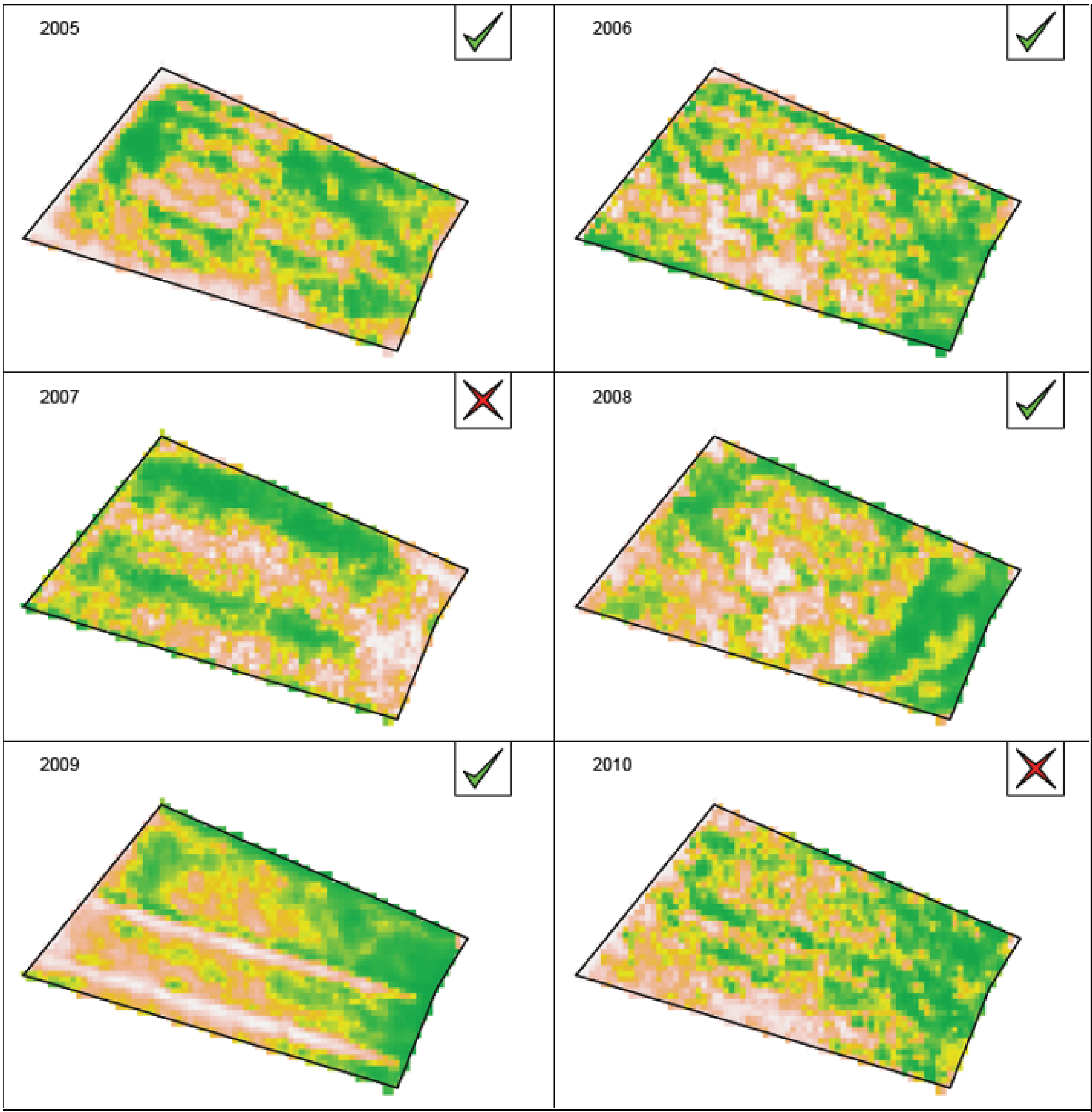


Figure 1. Maps of the index representing the spatial distribution of crop performance at Sunbury for the years 2005-2010. Green (darker areas) shows relatively good crop performance, beige relatively poor. The tick or cross in the corner of each panel indicates whether the analysis deemed that year to have been cropped or not.

Consistent spatial patterns in the remote-sensing data

It is important to note that the spatial pattern of the index has some aspects that repeat year after year, and other aspects that do not repeat. For example, in Figure 1 it seems that the east of the field was consistently higher yielding than the west. Also note that this was not the case in 2007, though for that year the field was deemed to not have been cropped. This highlights the importance of having a multiyear analysis when the goal is to identify soil constraints, which we would expect to give rise to similar spatial patterns of yield year after year (as opposed to diseases or weeds which might give reduced yields in part of the field in isolated seasons).

To identify the patterns that repeat season after season, the long-term average of the index is calculated for each pixel (using only the years when a crop was grown). A statistical test is then performed to evaluate whether there is evidence that a pixel consistently performs above or below average. Figure 2 shows an example of this type of analysis, from which it is clear that the east side of the field gave consistently better performance than the west side. Within some fields there will not be strong variation in soil constraints, and then we might not expect much consistent variation in yields. However, for fields where soil constraints exhibit strong within-field variation, we would expect a consistent pattern of spatial variation, such as that illustrated in Figure 2.

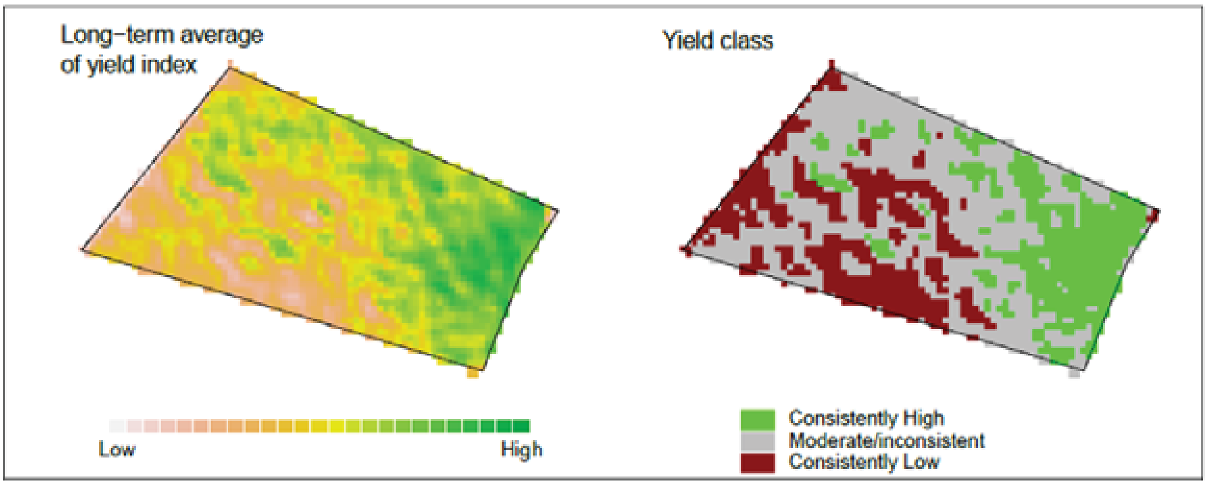


Figure 2. Maps summarising the multi-year analysis of remote-sensing data at Sunbury.

Comparing the remote-sensing information with soil data

The maps shown so far are based only on analysis of the remote-sensing data. Soil data from within the field are vital to validate what is causing the variation in yield. Information from the remote-sensing analysis (Figure 2) can be used to select locations for soil sampling. An advisable strategy might be to collect soil cores to a depth of 1.2 m from a few locations (minimum 3-4) in the areas with consistently high values of the crop yield index (green (mostly to the east) areas in Figure 2) and from a few locations in the consistently poor-performing areas (red (mostly to the west) areas in Figure 2). Once a dataset of soil analysis results from profiles in the field has been compiled, the software tool allows the user to upload/input the data, and it can be used to compare soil constraints in the in the good and poor-performing areas of the field.

For the Sunbury field, ten soil profiles were collected, of which two profiles were in the areas with consistently low crop yield index values, and three were in the consistently high yield index areas. The soil constraint data from the profiles in the better-performing areas are averaged and plotted as the green lines (generally to the left) in Figure 3; the red lines (generally to the right) show the averages from the poor-performing areas. Also shown are critical values for potential (light orange background - on left) or severe effects (light red background – on right) of the soil constraints on wheat yields. These critical values have come from a recent review of the literature on soil constraints to cropping in Australia (Page et al., 2020).

These plots indicate that there are differences in the salinity of the soil profiles from the constrained and unconstrained areas, with the profiles from the constrained areas exceeding the critical values for severe salinity effects below around 40cm in the soil profile. The differences between the constrained and unconstrained profiles in terms of sodicity were not as great.

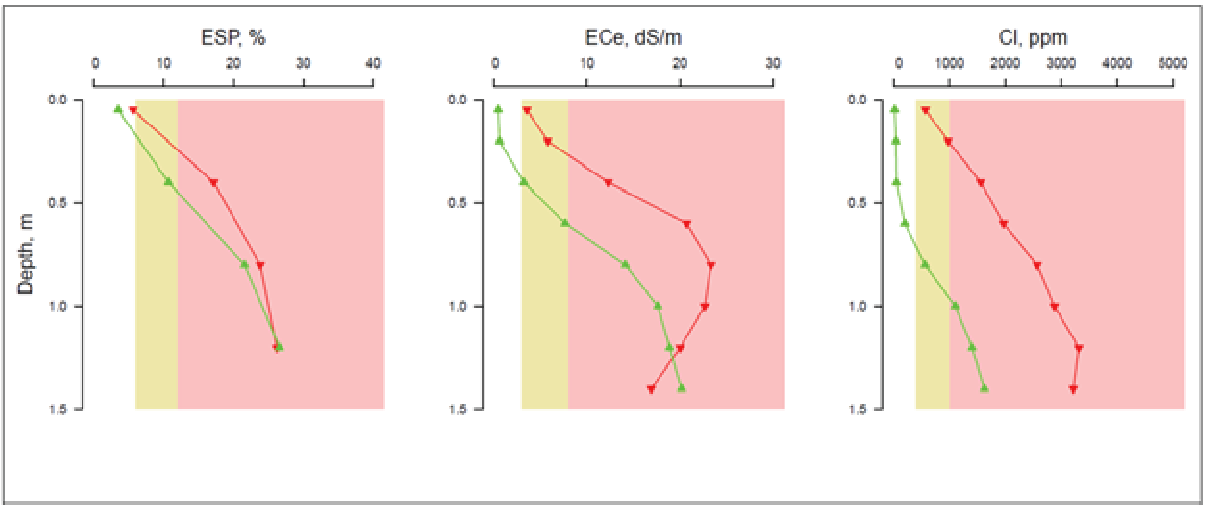


Figure 3. Soil profiles within the areas with consistently high crop yield index values (green - line to left) and consistently low values (red – line to right). More severely constrained zones are delineated by colour coding and are to the right side of each graph.

Web-based ConstraintID (http://constraintid.com.au/)

The web-based interface of Constraint ID allows farmers and consultants to setup, save and share soil constraint analyses for different paddocks (Figure 4).

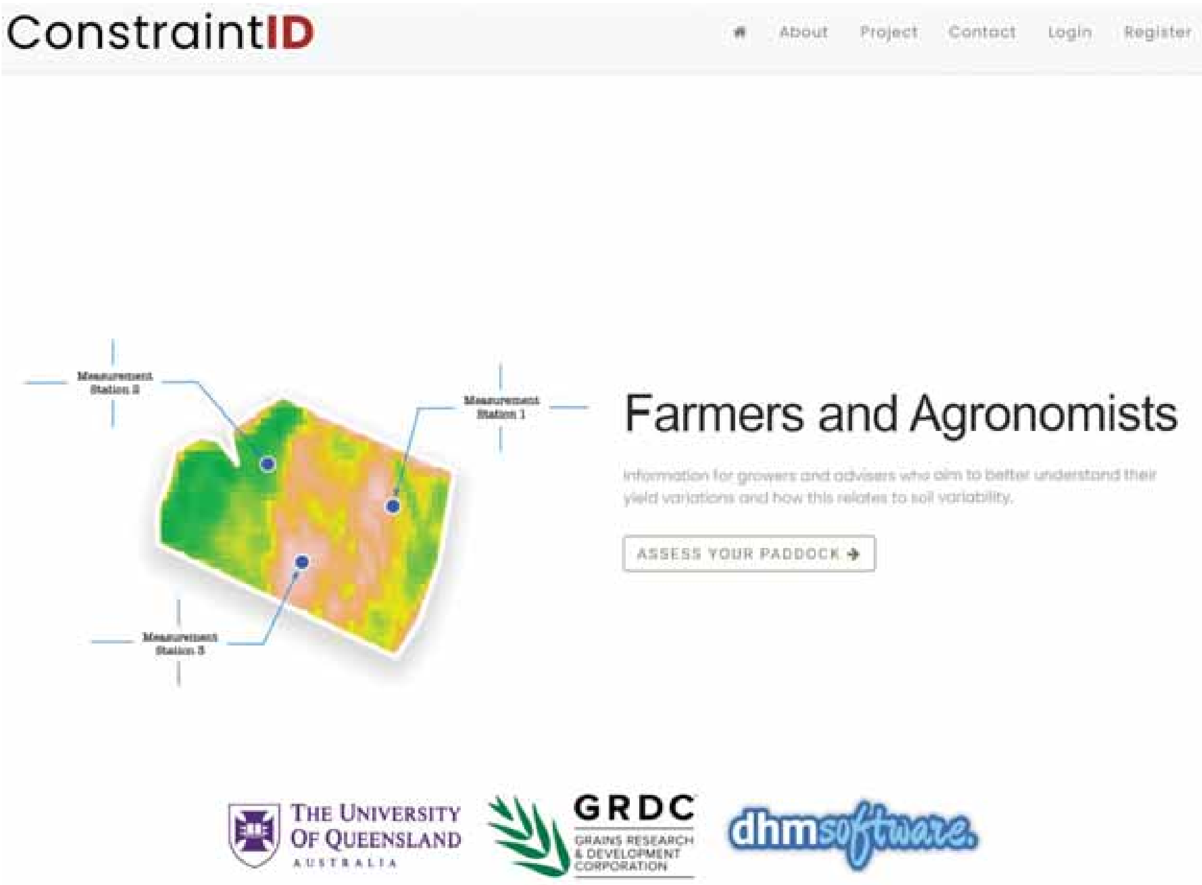
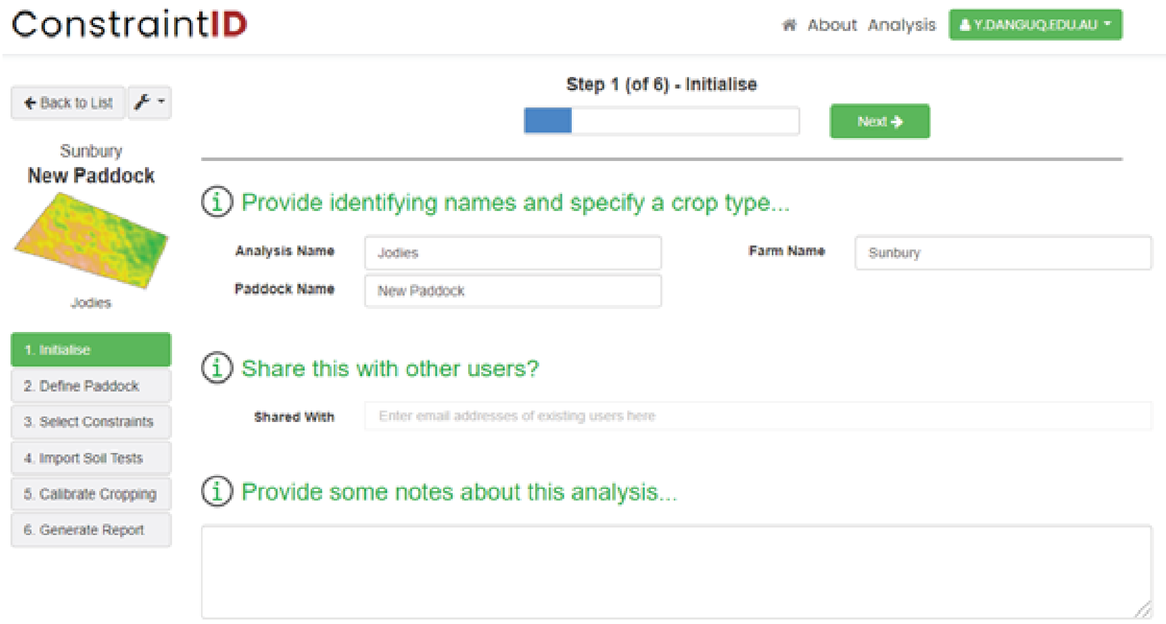


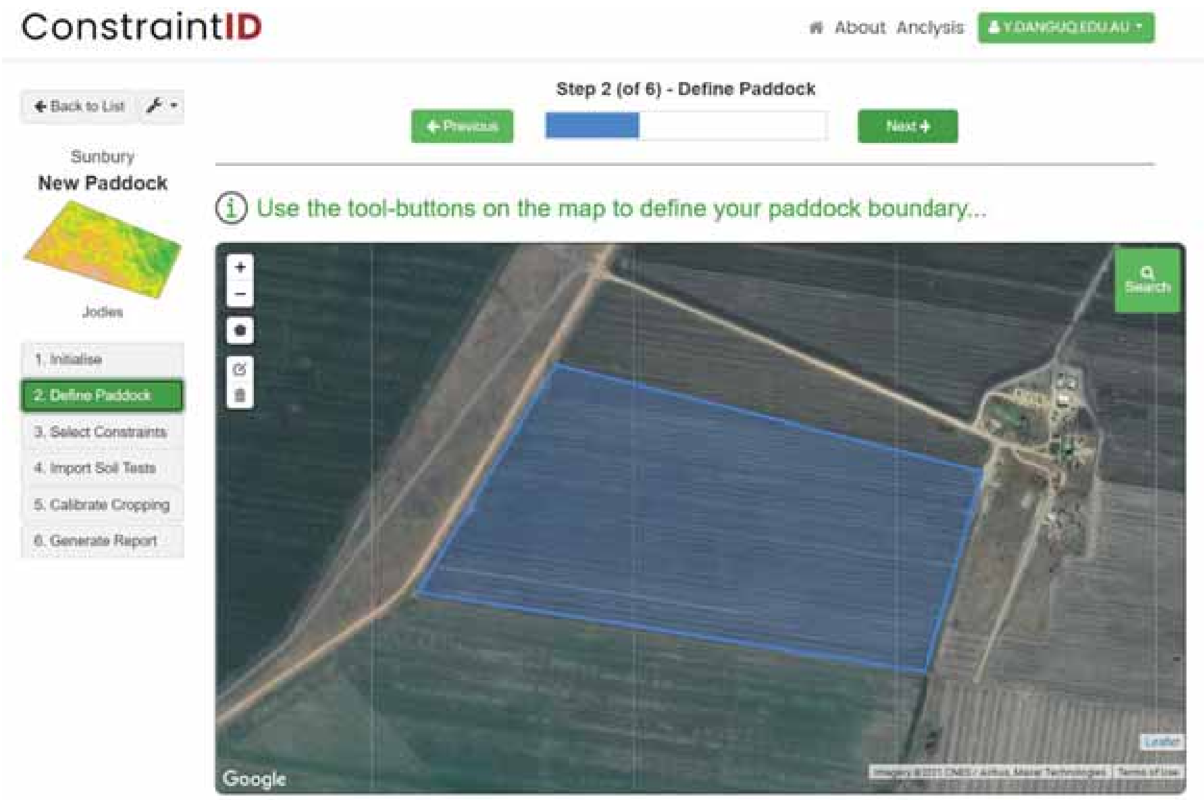
Figure 4. ConstraintID landing page

An analysis is broken down into 6 steps presented through the interface:

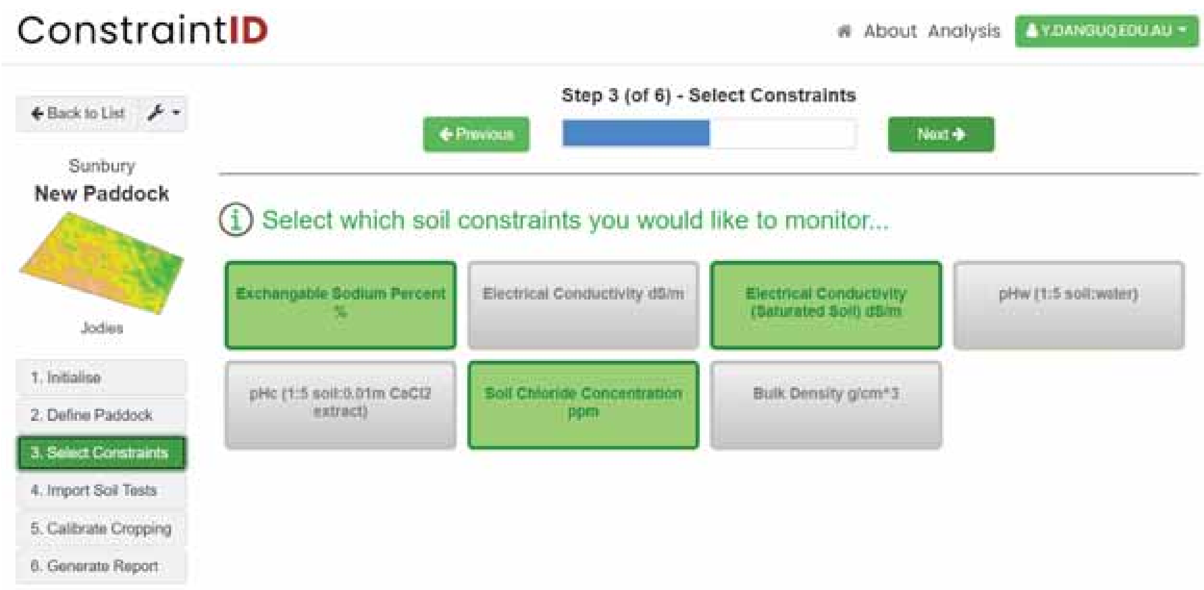
Step 1: Enter analysis details



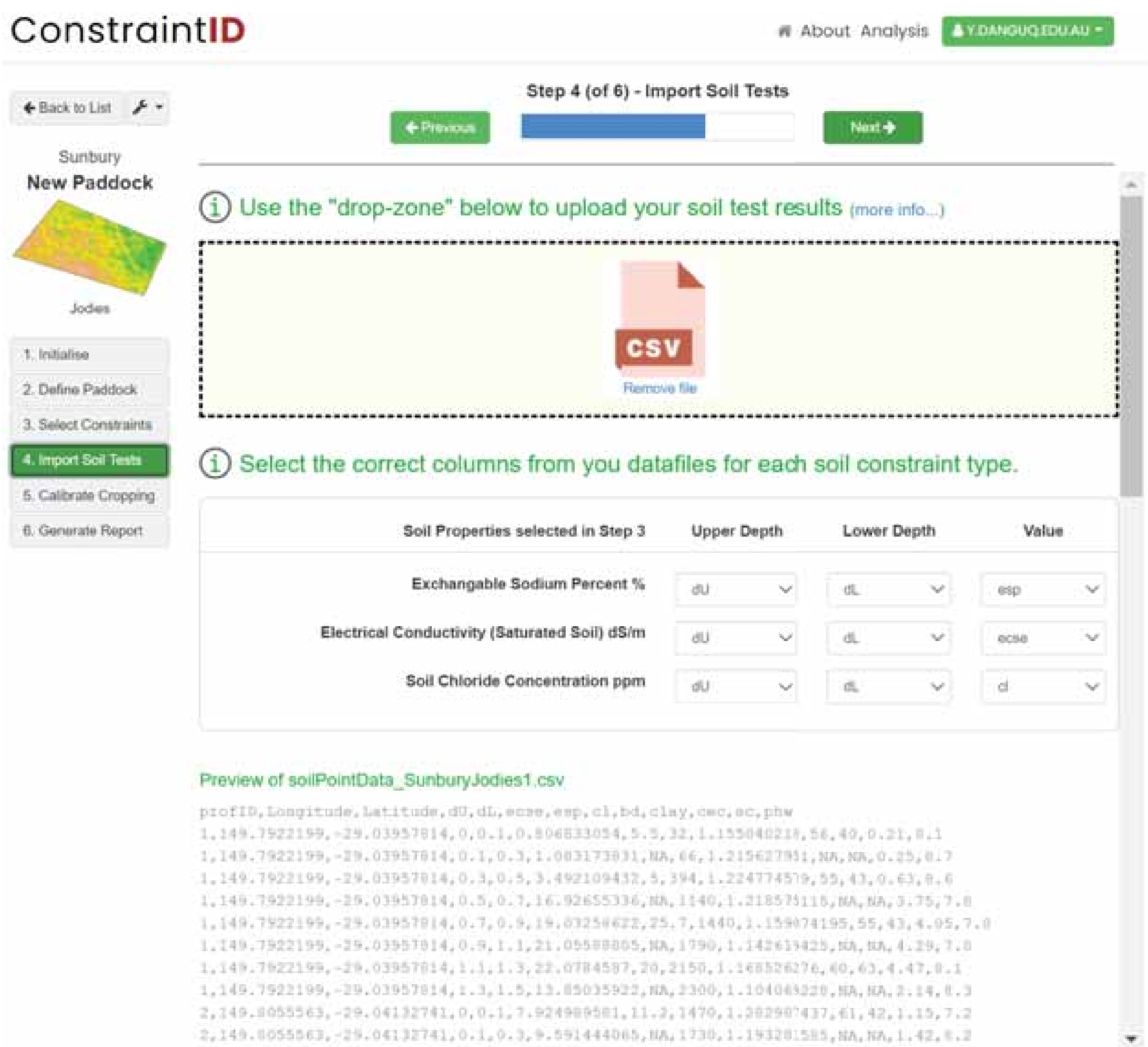
Step 2: Define paddock boundary



Step 3: Select soil constraints



Step 4: Import soil test data



Step 5: Select cropping years

Step 6: Review results

For each analysis, the software requires the user to trace their paddock boundary over a Google Maps' satellite image, using tools provided in Step 2. This then initiates the start of the spatial analysis which occurs silently in the background on the server. In this process, a time-series of 'Enhanced Vegetation Index (EVI)' satellite images from 1999 to the present date are extracted which typically include up to 20 images (days) per year. These tiled images are then reprocessed to exclude any data outside of the paddock boundary. In some cases, multiple adjoining images may require stitching together when the paddock boundary extends past the tile boundary. The resulting paddock images are then filtered to remove any that might be affected by excessive cloud cover or are indicative of fallow conditions. Finally, the individual EVI pixel values are ranked in order across the paddock to differentiate between high vegetation and low vegetation areas of the paddock. A single image is then produced for each year, based on averaging the ranked EVI values for that year, which we designate as the Crop Yield Index (CYI). This process typically takes 10-20 seconds to complete.

In the next steps (3 & 4), the user selects which soil constraints they wish to analyse and upload any measured soil test data they have relating to these constraints. Soil test data must be prepared in a comma-separated value format and uploaded to the server via dragging and dropping the data files into the user interface. Users must then identify which columns in the data files correspond to each soil constraint using in-built selection tools.

The second last step of the analysis involves the user being presented with a series of paddock images (generated in the second step) showing the Crop Yield Index across the paddock for each year. Each image is 'marked' to show whether or not the software thinks that that year was indicative of a typical cropping year. The user must then inspect each image to ensure that all typical cropping years are selected, and non-cropping years are de-selected. This in effect is calibrating the analysis in order to generate the final result.

In the final step of the analysis, all included cropping years are compiled into a single paddock map of Crop Yield Index values. A second image is then generated identifying those regions of the paddock that have demonstrated consistently high Crop Yield Index values (coloured blue) or consistently low values (coloured red). The locations of the soil test readings are then overlayed on these regions, allowing a graph of each soil constraint to be generated based on the soil test results, with different lines for high and low yielding areas.

Summary

The work presented here provides a snapshot of the Constraint ID software, which we think should provide growers with a valuable tool for investigating within-field variability and for making more informed management decisions. Further work to investigate and compare different management strategies once constrained areas have been identified is imperative.

References

Dang YP, Dalal RC, Buck SR, Harms B, Kelly R, Hochman Z, Schwenke GD, Biggs AJW, Ferguson NJ, Norrish S, Routley R, McDonald M, Hall C, Singh D K, Daniells I G, Farquharson R, Manning W, Speirs S, Grewal H S, Cornish P, Bodapati N, and Orange D (2010). Diagnosis, extent, impacts, and management of subsoil constraints in the northern grains cropping region of Australia. Soil Research, 48(2), 105.

Orton TG, Mallawaarachchi, T P, Matthew J, Menzies NW, Dalal RC, Kopittke PM, Searle R, Hochman Z and Dang YP (2018). Quantifying the economic impact of soil constraints on Australian agriculture: A case-study of wheat. Land Degradation & Development, 29(11), 3866–3875.

Page KL, Dang YP, Dalal RC, Kopittke PM and Menzies NW (2020). The impact, identification and management of dispersive soils in rainfed cropping systems. European Journal of Soil Science (accepted).

Acknowledgements

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.

Contact details

Dr Yash Dang
School of Agriculture and Food Sciences
203 Tor Street, Toowoomba
Ph: 0427 602 099
Email: y.dang@uq.edu.au

Amelioration for sodicity - deep ripping and soil amendment addition across NSW and Qld. Engineering challenges. Yield responses to ripping, gypsum and OM placement in constrained soils

David Lester[1], Cameron Silburn[2], Craig Birchall[3], Richard Flavel[3], Chris Guppy[3], John Maclean Bennett[4], Stirling Roberton[4], David McKenzie[5]

[1] Department of Agriculture and Fisheries Queensland, Toowoomba
[2] Department of Agriculture and Fisheries Queensland, Goondiwindi
[3] University of New England, Armidale
[4] University of Southern Queensland, Toowoomba
[5] Soil Management Designs, Orange

Key words

yield maps, soil testing, ground truthing, dispersion, sodicity, ripping

GRDC code

USQ1803-002RTX

Take home messages

- Benefits of subsoil amendments are more likely in poorer seasons
- Adjustment in agronomy may be necessary to take advantage of improved subsoil conditions
- Considerable challenges remain in identifying engineering solutions to deeper amendment applications.

Background

Model analyses suggest a yield gap between water-limited potential yield and currently achieved production exists across northern Australian grain regions. This yield gap is a function of physical, chemical and biological factors in each soil, including capacity of soil to store and release water for efficient plant use. Many regions where yield is constrained contain dispersive soil within the surface 50cm and deeper. Sodicity (a high exchangeable sodium percentage) is a major cause of aggregate dispersion and may compromise soil structure. Dispersive behaviour decreases both soil water availability and nutrient acquisition, increases risk of runoff and erosion, and impairs biological (soil microbial and plant root) activity. Acidity, salinity (presence AND absence) and compaction further constrain yield potentials. However this project focuses on sodicity as the major constraint with often related constraints considered as compounding and/or interacting factors.

Amelioration of subsoil constraints is an expensive process. The engineering challenges and energy requirements are significant. It is important to acknowledge that production benefits from subsoil amendment are more likely to be observed in poorer seasons. In good seasons, root function and activity, and soil moisture, will be able to sustain yield from surface activity and extraction where soils are often less constrained. However in poorer seasons where subsoil moisture is required to finish a crop, subsoil amelioration will have a proportionately larger impact on yield. Hence, we expect that the return on investment in subsoil amelioration to be larger in poorer seasons, than in higher decile seasons with more growing season rainfall.

A series of linked investments is assessing the economics of ameliorating constrained surface and sub-surface soils in the northern region. The program has four areas covering:

(i) Spatial soil constraint identification
(ii) Amelioration and management of soil constraints

(iii) Economics of adoption, and
(iv) An overarching communications and extension program.

The research into soil amelioration and management has two components led by the University of Southern Queensland (USQ). First is a set of six small-plot core experiments exploring detailed amelioration research. There are three sites in northern and central New South Wales (NSW) managed by the University of New England (UNE), and three sites in southern Qld managed by the Department of Agriculture and Fisheries (DAF).

This paper covers the challenges of core site implementation, describes the treatments being studied and the adaptations needed to deliver these treatments to depth in our constrained soils. It also extends to discussing the realities of attempting this process at a commercial scale. Finally it reports on the first season of field trial responses.

Core site selection

The USQ team undertook extensive surveying of 30 fields across central and northern NSW and southern Qld. The areas were clustered around 6 locations (Trundle, Armatree, Spring Ridge, Talwood, Millmerran and Meandarra), with 5 fields surveyed at each. Field surveying included capture of yield maps if available, satellite NDVI imagery, and soil mapping with EMI. Using a combination of yield, site elevation and EMI maps, bare soil colour imagery and grower experience, four survey points where selected for soil sample collection and analysis. After consolidating data 'core' experimental sites where selected in each of the six hub areas: three in NSW and three in Qld.

Core site characterisation

All sites were generally alkaline in the upper profile with an exchangeable sodium percentage (ESP) well over the 6% nominal threshold for healthy crop growth. Profile chloride (Cl) values were generally low, indicating that sodicity was likely to be the primary restriction.

Chemical characteristics of the six core experiments are below:

Location: Armatree
Soil type: Brown Sodosol, not dispersive (0-10cm) to dispersive (10-20) surface, to strongly alkaline and dispersive at depth, compact surface layers

Depth	pH	pH	EC	Ca	Mg	Na	K	ECEC	ESP	Cl	P
(cm)	(H_2O)	($CaCl_2$)	(1:5)	(cmol/kg)					%	(mg/kg)	(mg/kg)
0-10	6.0	5.3	0.20	3.7	3.4	0.91	1.00	9.0	10		58
10-20	7.8	6.8	0.20	8.5	7.9	2.63	0.83	19.9	13		7
30-40	9.3	8.3	0.45	13.1	12.6	5.78	0.81	32.3	18		8
60-70	9.4	8.4	0.58	12.3	13.4	6.35	0.96	33.0	19		6

Location: Forbes
Soil type: Brown Vertosol, not dispersive (0-10cm) to dispersive (10-20) surface, to strongly alkaline and dispersive at depth

Depth	pH	pH	EC	Ca	Mg	Na	K	ECEC	ESP	Cl	P
(cm)	(H_2O)	($CaCl_2$)	(1:5)	(cmol/kg)					%	(mg/kg)	(mg/kg)
0-10	6.3	6.1	0.39	8.7	7.7	2.13	0.77	19.3	11		89
10-20	7.9	6.9	0.30	15.4	10.2	4.76	0.55	30.9	15		12
30-40	9.1	8.2	0.64	12.5	11.3	8.14	0.49	32.5	25		4
60-70	9.1	8.3	0.85	11.3	10.4	9.57	0.56	31.9	30		1

Location: Spring Ridge
Soil type: Black Vertosol, moderate ESP and salinity in surface, increasing to high ESP and salinity at depth, but both are non-dispersive due to the salinity

Depth	pH	pH	EC	Ca	Mg	Na	K	ECEC	ESP	Cl	P
(cm)	(H_2O)	($CaCl_2$)	(1:5)	(cmol/kg)					%	(mg/kg)	(mg/kg)
0-10	8.2		0.54	31.7	41.7	3.3	2.4	79.1	4		100
10-20	8.2		0.62	37.2	43.5	5.2	1.4	87.3	6		
30-40	8.3		1.94	31.0	51.5	13.9	1.0	97.4	14		
60-70	8.3		2.52	28.7	56.7	19.5	1.1	106	18		

Location: Dulacca
Soil type: Grey/Brown Vertosol (nominally Ulimaroa). Surface soils not spontaneously dispersive, subsurface highly dispersive.

Depth	pH	pH	EC	Ca	Mg	Na	K	ECEC	ESP	Cl	P
(cm)	(H_2O)	($CaCl_2$)	(1:5)	(cmol/kg)					%	(mg/kg)	(mg/kg)
0-10	8.5	7.7	0.21	18.1	8.0	2.73	0.93	29.8	9	43	9
10-20	8.8	7.8	0.25	15.8	9.8	3.99	0.61	30.3	13	53	14
30-40	8.1	7.3	0.46	15.4	12.3	7.10	0.45	35.3	20	102	4
60-70	6.8	6.7	0.66	12.0	12.8	8.83	0.48	34.1	26	275	8

Location: Millmerran
Soil type: Grey/Brown Vertosol (nominally Moola). Surface and subsurface soils not spontaneously dispersive, very compact soil through the profile.

Depth	pH	pH	EC	Ca	Mg	Na	K	ECEC	ESP	Cl	P
(cm)	(H_2O)	($CaCl_2$)	(1:5)	(cmol/kg)					%	(mg/kg)	(mg/kg)
0-10	6.6	6.3	0.15	8.4	6.6	2.37	0.31	17.7	13	153	38
10-20	8.7	7.4	0.24	10.6	9.0	3.36	0.20	23.2	14	330	5
30-40	6.9	6.2	0.38	9.5	15.	6.82	0.14	31.4	22	428	3
60-70	6.4	5.5	0.43	10.2	16.4	8.79	0.18	35.5	25	457	2

Location: Talwood
Soil type: Red/Brown Vertosol (nominally Arden). Surface soils not spontaneously dispersive, subsurface highly dispersive at 60-70cm.

Depth	pH	pH	EC	Ca	Mg	Na	K	ECEC	ESP	Cl	P
(cm)	(H_2O)	($CaCl_2$)	(1:5)	(cmol/kg)					%	(mg/kg)	(mg/kg)
0-10	8.3	7.6	0.17	27.5	4.7	1.8	1.3	35.5	11	22	18
10-20	8.7	7.9	0.23	27.8	7.0	3.8	0.7	39.3	10	26	3
30-40	8.9	7.8	0.36	22.5	9.4	7.0	0.4	39.4	18	73	2
60-70	9.2	7.9	0.44	20.3	9.9	9.9	0.5	40.7	24	163	2

Experiment treatments

This research focussed on eliminating sodium as a constraint for the upper 50 cm of a soil profile. It was 'proof-of-concept' research, intended to explore effects on soil water storage and grain yields under gypsum application rates to remediate the ESP to ≤ 3% in either or both of the top 20 cm of soil and half of the soil volume in bands from 20 cm down to 50 cm depth. Organic matter (OM) also acts to limit aggregate dispersion (as well as providing nutrients at depth) and, whilst not reducing ESP, may act to improve water holding capacity and pore stability. Gypsum rates (often ≥ 15 t/ha) were compared against a high rate subsoil (≈ 20 cm deep) compost (Qld)/lucerne pellet (NSW) application (≈ 10 t/ha), and the application of elemental sulfur to dissolve calcium carbonate to produce gypsum in-situ and lower alkalinity.

The rates chosen are considerably higher than the likely economically viable rates and were deliberately chosen to determine if subsoil remediation to remove dispersive constraints would result in improved production outcomes beyond the first year. With that in mind, it is worth considering that the cost in diesel of ripping to depth without adding the necessary amendment is unlikely to be recovered. Repeated, smaller gypsum/OM applications coupled with deep ripping to place them is cost prohibitive. Hence, single, large, additions may ultimately be best practice.

Similar treatment structures are used in both NSW and Qld, with both physical and chemical ameliorants, a range of options exploring impacts and/or interactions between tillage (shallow and deep), deep placement of nutrients (as inorganic or organic forms), surface and subsurface applications of gypsum to reduce ESP to < 3%, incorporating organic amendments (lucerne pellets in NSW and composted feedlot manure in Qld), and applying elemental sulfur (ES) to decrease soil pH and generate gypsum in situ (Table 1).

Surface gypsum treatments were spread onto the soil, and then incorporated by ripping to 20 cm. Actual application rates for gypsum varied with each site based on calculations that capture the required Ca to lower the ESP to <3%, but the overall structure of the experiment stayed the same.

The applied gypsum rate for subsurface placement was banded with 50% of the total needed for the whole 20 to 50 cm layer of soil. For example, if a total of 20 t/ha of gypsum was theoretically needed to remediate the entire 20 to 50 cm layer, in this application 10 t/ha was applied to ensure the right amount of gypsum within each band (assuming the band covers 50% if this zone). In NSW, it was assumed that the band only treated 25% of the profile, so only 25% of the total gypsum needed was applied – 5 t/ha in the example. Further details on how it was applied are outlined later in this paper.

Table 1. Treatment structure for core soil constraints sites in southern Queensland

Treatment	Rip (≈ 20 cm)	Surface Gyp	Deep NP(K)Zn (≈ 20 cm)	Rip > 20 cm	Deep Gyp (≈ 20 cm)	Deep OM (≈ 20 cm)	Deep ES (≈ 20 cm)
1							
2	Y						
3	Y		Y				
4	Y	Y	Y				
5	Y		Y	Y			
6	Y		Y	Y	Y		
7	Y	Y	Y	Y	Y		
8	Y	Y	Y				
9	Y	Y	Y	Y			Y
10	Y	Y	Y (*)	Y			
11	Y	Y		Y		Y	
12	Y	Y		Y		Y	Y
13	Y	Y		Y	Y	Y	Y

Deep NP(K)Zn rate is 50 kg N, 30 kg P, 50 kg K and 1.5 kg Zn apart from * rate which matches N and P addition from deep compost application. Gyp = Gypsum, OM = Organic matter ES = elemental sulfur

Engineering and application challenges

Application rates for all ameliorants are substantial, with rates often greater than 6-10 t/ha being applied. With such large amounts of product required, logistically it is challenging to source, transport and apply the treatments at each site, especially with deep (~20 cm) applications.

Applicators are not going to be universally capable machines for putting ameliorant at depth – it is very challenging to engineer something to measure and distribute all manner of materials. The deep placement machine used in Qld required amendments with the physical characteristics of being relatively fine and easily flowable in order to be metered and delivered to depth. This was achieved by using 6 mm screened feedlot compost, and air drying natural mined gypsum to reduce moisture content. All materials were screened at the top of the applicator bin through a 12mm screen to further exclude lumps. In NSW, flow problems were resolved by using pellets and prills – a more expensive solution.

Metering of products was achieved through fluted rollers (Figure 1). Further changes were undertaken adapting it for use in the soil amendment experiments. Modifications included 75 mm chutes for amendment delivery down the back of the applicator tine and inclusion of a crumble roller for smoothing the surface slightly.

Figure 1. Metering unit for amendment application on the Qld research machine, with ruler for scale.

Given the 'proof-of-concept' intent of much of the research, interpretation of individual specific effects is challenging. What the research outcomes do confirm is there exists a significant upside in yield achievable with combinations of physical and chemical inputs.

Grain yield

Millmerran was planted to sorghum for 2019-20, while Dulacca, Armatree, Forbes and Spring Ridge all had winter crops in 2020. While experiments are similar across the Qld and NSW components, the results for the different states are reported separately.

Grain yield responses in Qld

In Qld, yield increases have been recorded at both the harvested sites. At Millmerran (Table 2), grain yields increased up to ≈25% or 750 kg/ha. In general, treatments with combinations of surface gypsum and subsurface NPK (i.e. trt 4, 7, 8 and 13) had the largest yield gains.

Table 2. 2019-20 Grain yield (GY) for sorghum at Millmerran

Trt		Yield	SE	Delta GY	Rel GY%	LSD*	Protein
1	Control	2970	133	0	0	a	11.1
2	Shallow rip	2910	188	-60	-2	a	10.7
3	Banded fert	3300	188	330	11	abcd	10.7
4	Surface gyp + shallow rip	3580	133	610	21	bcd	10.4
5	Deep rip	3430	188	460	15	abcd	10.6
6	Deep gyp	3120	188	150	5	ab	10.9
7	Surface + deep gyp	3750	133	780	26	d	10.7
8	Surface gyp + deep rip	3750	188	780	26	cd	10.2
9	ES + surf gyp	3250	188	280	10	abc	10.6
10	Nutrient control	3500	188	530	18	bcd	11.0
11	Surface + deep OM	3370	188	400	13	abcd	10.4
12	ES+OM	3360	188	390	13	abcd	10.5
13	All	3700	188	730	25	cd	10.5

* Treatments with the same letter do not significantly differ (P<0.05)

At Drillham (Table 3), grain yields increased up to ≈60% or 1,250 kg/ha. Three treatments in particular provided the largest increases, treatments 10, 12 and 13. Commonality of these treatments is all have tillage to 30 cm, and either have high nutrient supplies from the high NP rates (treatment 10), or the composted feedlot manure (12 and 13). Deeper ripping (to 30cm) and lower nutrient inputs (50N,30P) increased yields by 800 kg/ha (40%), but it appears the higher nutrient supply plots allowed greater yields to be recorded.

Table 3. 2020 Grain yield (GY) for wheat at Drillham

Trt		Yield	SE	Delta GY	Rel GY%	LSD*	Protein
1	Control	2110	66	0	0	a	13.7
2	Shallow rip	2200	133	90	5	ab	13.5
3	Banded fert	2520	133	410	20	bcd	14.4
4	Surface gyp + shallow rip	2320	133	210	10	abc	14.0
5	Deep rip	2990	133	880	42	fgh	13.3
6	Deep gyp	2860	133	750	35	defg	13.8
7	Surface + deep gyp	2580	133	470	22	bcde	13.8
8	Surface gyp + deep rip	2910	133	800	38	efg	13.8
9	ES + surface gyp	2690	133	580	27	cdef	13.5
10	Nutrient control	3310	133	1200	57	hi	13.9
11	Surface +deep OM	3010	133	900	43	fgh	13.7
12	ES + OM	3200	133	1090	52	ghi	13.6
13	All	3420	133	1310	62	i	13.6

* Treatments with the same letter do not significantly differ (P<0.05)

Grain responses in NSW

The Armatree site produced a 4.5t/ha crop at around 15% protein and had significant differences between treatments in both grain yield, and biomass at flowering (Table 4). In general, deep ripping and the addition of nutrients, as banded fertiliser or contained in the organic amendment, increased growth and yields by approximately 20%. However, the organic matter treatments (11-13) ran out of water during grain fill due to the very large biomass produced, resulting in similar yields to the controls, with higher protein and screenings levels.

Table 4. 2020 Grain yield (GY) for wheat at Armatree

Trt		Yield	SE	Delta GY	Rel GY%	LSD*	Protein	Screenings
1	Control	4470	188	0	0	abcd	15.5	0.8
2	Shallow rip	4740	210	270	6	abcde	15.8	1.2
3	Banded fert	5040	210	570	13	abcde	14.8	0.9
4	Surface gyp + shallow rip	5210	210	740	17	bcde	15.4	1.2
5	Deep rip	5390	210	920	21	cde	14.7	1.1
6	Deep gyp	5460	210	990	22	cde	15.0	1.1
7	Surface + deep gyp	5110	188	640	14	bcde	14.6	1.2
8	Surface gyp + deep rip	5330	188	860	19	cde	15.0	1.0
9	ES + surface gyp	5630	210	1160	26	e	14.9	1.0
10	Nutrient control	5340	210	870	19	bcde	14.8	1.1
11	Surface + deep OM	4390	210	-80	-2	abc	16.3	2.1
12	ES + OM	4570	210	100	2	abcde	17.0	3.1
13	All	4340	188	-130	-3	ab	17.1	2.2

* Treatments with the same letter do not significantly differ (P<0.05)

Forbes had no significant differences in canola grain yield, but the organic treatments did increase protein and reduce oil content (Table 5). The site was waterlogged for most of winter, and still had moisture in the profile at harvest, so any differences in plant available water capacity, or root penetration to depth, were unlikely to have shown up in this season. The plant population was also extremely variable due to the surface roughness from the ripping and waterlogging from 70mm of rain immediately after sowing. This variation has been included in the statistical analysis, but also suggests that small-seeded canola may not be the best crop for growing immediately after a ripping program.

Table 5. 2020 Grain yield (GY) for canola at Forbes

Trt		Yield	SE	Delta GY	Rel GY%	LSD*	Protein	Oil
1	Control	2470	194	0	0	ns	19.9	44.9
2	Shallow rip	2530	222	60	2	ns	19.9	44.3
3	Banded fert	3070	193	600	24	ns	20.3	43.9
4	Surface gyp + shallow rip	2710	192	240	10	ns	20.4	45.0
5	Deep rip	2680	176	210	9	ns	20.8	43.2
6	Deep gyp	2650	193	180	7	ns	21.4	42.7
7	Surface + deep gyp	3010	172	540	22	ns	20.6	43.7
8	Surface gyp + deep rip	2840	192	370	15	ns	20.4	44.0
9	ES + surf gyp	2700	193	230	9	ns	20.4	43.7
10	Nutrient control	2750	193	280	11	ns	20.2	44.2
11	Surface + deep OM	2630	173	160	6	ns	22.4	40.6
12	ES + OM	2570	192	100	4	ns	22.5	40.4
13	All	2590	192	120	5	ns	21.6	41.7

* Treatments with the same letter do not significantly differ (P<0.05). ns = not significant difference

Spring Ridge also had no significant differences in biomass at flowering, or barley grain yield (Table 6). The high yield reflects the good season in 2020 and suggests that the site may not be as constrained as originally thought. We are uncertain if this lack of observed yield constraint is long term, or due to the above average rainfall in early 2020 which may have resulted in short term leaching of the salinity found in the initial sampling.

Table 6. 2020 grain yield (GY) for barley at Spring Ridge

Trt		Yield	SE	Delta GY	Rel GY%	LSD*
1	Control	6520	226	0	0	ns
2	Shallow rip	6030	253	-490	-8	ns
3	Banded fert	6170	256	-350	-5	ns
4	Surface gyp + shallow rip	6410	254	-110	-2	ns
5	Deep rip	6320	256	-200	-3	ns
6	Deep gyp	6360	256	-160	-2	ns
7	Surface + deep gyp	6700	227	180	3	ns
8	Surface gyp + deep rip	6050	227	-470	-7	ns
9	ES + surf gyp	5760	256	-760	-12	ns
10	Nutrient control	6460	254	-60	-1	ns
11	Surface + deep OM	6170	256	-350	-5	ns
12	ES + OM	6270	254	-250	-4	ns
13	All	6310	227	-210	-3	ns

* Treatments with the same letter do not significantly differ ($P<0.05$) ns = no significant difference.

Conclusions

We present one year of results, after a very wet summer/autumn which allowed all plots to refill after treatment application except for the Talwood site which has been planted in mid-January 2021. The 2021 crop will give a better indication of long term effects of treatments, and (possibly) under more typical conditions. Increases in yield as a result of the ripping may also require extra nutrients to achieve the increased yield potential.

Acknowledgements

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 authors would like to thank them for their continued support.

Contact details

David Lester
Department of Agriculture and Fisheries Queensland, Toowoomba
PO Box 2282, Toowoomba, Qld 4350
Ph: 07 4529 1386
Email: david.lester@daf.qld.edu.au

Chris Guppy
University of New England, Armidale
Agronomy and Soil Science
Ph: 02 6773 3567
Email: cguppy@une.edu.au

Identification of crop –specific and varietal tolerance limits to acidity, salinity and sodicity

Jack Christopher[1], Kathryn Page[2], Yash Dang[2] and Neal Menzies[2, 1]

[1] Queensland Alliance for Agricultural and Food Innovation, The University of Queensland
[2] School of Agriculture and Food Sciences, The University of Queensland

Key words

soil constraints, chloride toxicity, variety selection, proxy traits

GRDC codes

UA000159: Improving wheat yields on sodic, magnesic and dispersive soils

UOQ1803-003RTX: Economics of ameliorating soil constraints in the northern region: Spatial soil constraint diagnoses

Take home messages

- In Qld and NSW, the soil constraints affecting the greatest cropping areas are sodicity combined with toxic chloride (Cl) concentrations. Acid sub-soils are also important in some areas
- Reduced ability to extract soil water from deeper layers is the main affect reducing yield
- Crop-specific tolerance limits vary between species and between soil constraints
- Tolerance to soil constraints differs between varieties of wheat
- Robust ranking would allow growers and breeders to select the most tolerant varieties, but high spatial soil variation over short distances makes consistent ranking difficult
- We are testing whether pre-screening for proxy traits in the laboratory and using remote sensing to account for spatial variation in the field can be used to increase our ability to select the most tolerant varieties.

Introduction

Successful dryland crop production in the north-eastern Australia depends on utilising soil moisture accumulated in the period preceding sowing. Due to the high clay content of the soils in the region, these soils can potentially store 200-250 mm of water in the soil profile or more. However, soil constraints, especially in the subsoil, reduce the effective rooting depth thus also reducing the amount of water and nutrients that plants can obtain from the soil, resulting in reduced crop yield (e.g. Figure 1) (Dang et al. 2006 and 2016).

High sodicity in surface soil and subsoil, high salinity and phytotoxic concentration of chloride (Cl) in the subsoil, alkalinity, subsoil acidity and compaction are common soil constraints in many semi-arid regions worldwide and in particular Queensland and NSW (Figure 2, Table 1). These soil constraints often occur in combination and can interact with each other to create unique environments for root growth at a given location (Nuttall et al. 2003). They may also vary both spatially and temporally. Spatial variation can occur within a field, across the landscape and with depth in the soil profile.

Spatial variability and the interactions between constraints can limit agronomic and management options. Variability in the impact on crop growth and yield is also compounded by the interaction between constraints and environmental factors, particularly the timing and amount of rainfall relative to the crop development cycle.

The amelioration of soils affected by constraints is usually expensive, if it is possible at all. The application of ameliorants (e.g. gypsum for sodicity) can sometimes improve surface soils (Page et al 2020, 2021). However, amelioration of constraints occurring in the subsoil is not usually practical.

Where subsoil constraints are present, the selection of crops or varieties tolerant to soil constraints and the identification of traits for pre-selection may provide a long-lasting, tangible solution to improve yields.

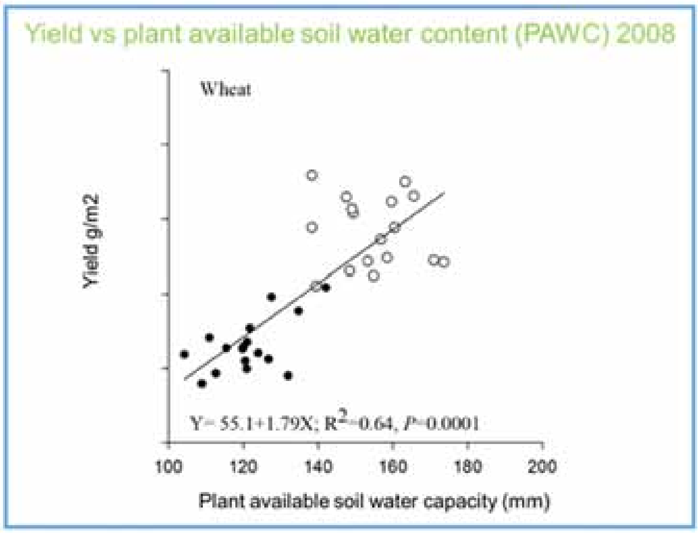


Figure 1. Wheat yield versus soil water extracted between sowing and maturity (plant available soil water capacity, Dang et al., 2016). Solid dots represent data for a highly sodic site and open dots a less-sodic site.

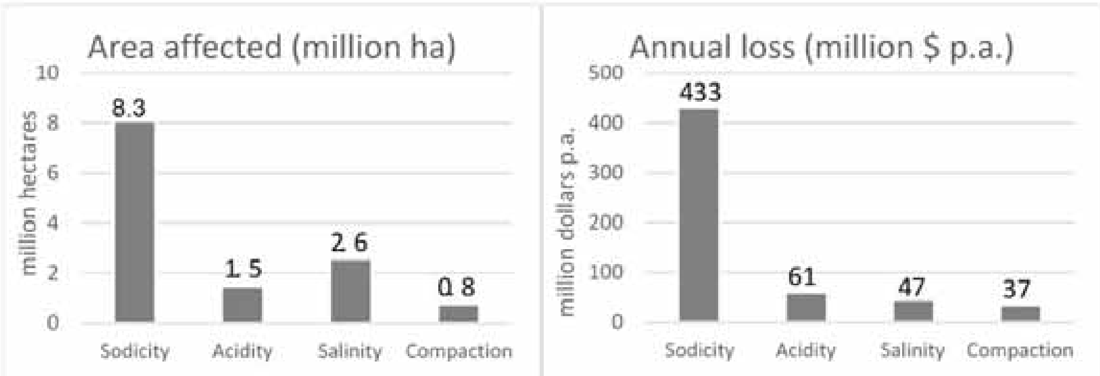


Figure 2. Area affected and annual financial loss due various soil constraints in Queensland and NSW (Orton et al. 2018)

Table 1. Sodicity combined with high chloride concentration at depth represent the main soil constraints in QLD and NSW (Orton et al. 2018).

Region	**Area of cropping land affected (million ha)**		**Estimated cost in lost production**
	Sodicity	Subsoil Cl	(million $/yr)
Central Qld	1.4	0.7	59.5
Southern Qld	1.4	0.9	85.4
Northern Qld	3.0	1.0	157.0
Central NSW	1.3	<0.1	54.3
Southern NSW	1.0	<0.1	77.0
Total	**8.1**	**2.7**	**433.1**

Crop-specific tolerance limits vary between species and between constraints

Saline soils

Soil salinity refers to the amount of salt dissolved in the soil solution and is quantified by measuring the electrical conductivity of a saturated soil extract (ECe). Lower ECe values are generally more favourable for crop production.

A recent survey of published studies on the tolerance of 17 crop species reported that the tolerance thresholds for soil constraints varied between species and between constraints (Page et al., 2021). The tolerance levels of different crops for soil salinity were classified using a 'traffic light' system (Figure 3). Salinity levels for which little if any reduction in yield was reported in the literature were classified green. Levels where some yield loss might be observed (depending on soil type, season and variety) are represented in yellow, while those where loss was reported for most soils, seasons and varieties are represented in red. For example, the studies indicate that the more tolerant cereals, such as barley and millet, will typically perform well up to salinity levels of ECe 5 ds/m. However, they usually suffer severe damage at levels above ECe 10 ds/m (Figure 3). In contrast, more sensitive cereals, such as maize and sorghum, only performed well up ECe 2 ds/m and suffered severe damage at levels greater than ECe 5 ds/m. Some legume species were even more sensitive (Figure 3).

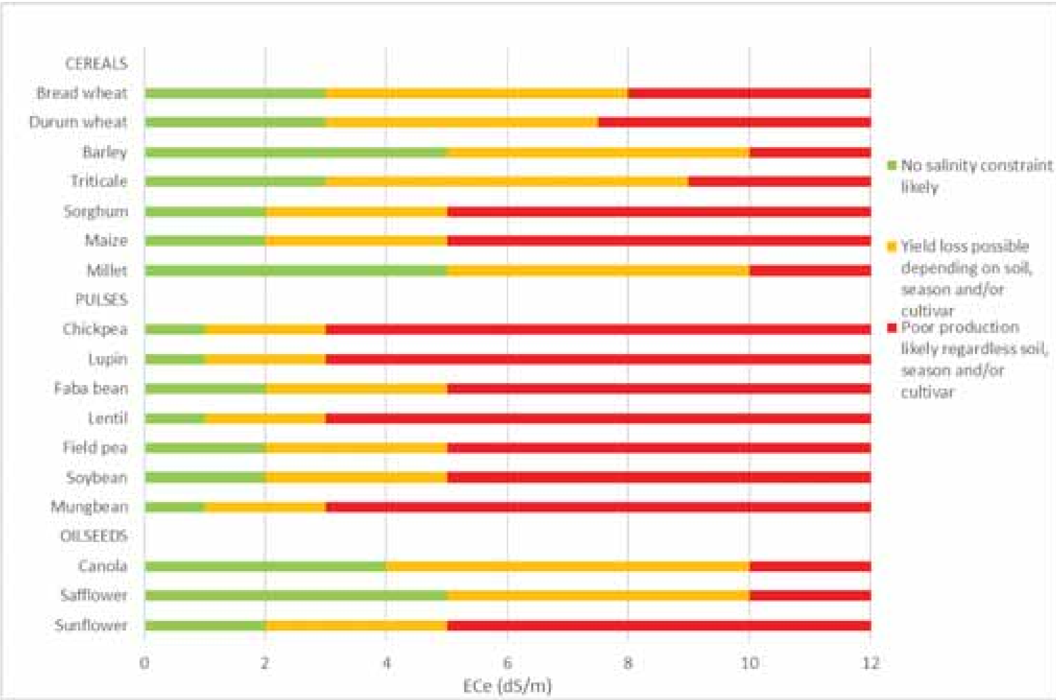


Figure 3. Soil EC_e tolerance limits proposed for crop species when grown under rainfed conditions in subtropical regions (Page et al., 2021).

Acid soils

The adverse effects of soil acidity generally increase with lower pH values measured in the soil solution.

Just as for salinity, the tolerance thresholds of crops varied between the 17 species and between constraints (Page et al., 2021). However, the ranking of the crops was very different. Results are represented here in Figure 4, again using the 'traffic light' system described above. For acidity, the cereals sorghum, maize and millet were able to tolerate more severe acidity levels than many other species, with good growth to as low as pH 5.5 and severe yield loss once pH dropped below pH 4.8.

In contrast, the most sensitive cereals were durum and barley, which performed well only down to pH 6.0 and suffered severe yield loss below pH 5.3. Some legume species were also similarly sensitive (Figure 4).

Thus, the crop species most suitable for a particular paddock can vary greatly depending on which are the most prevalent soil constraints. Crop and cultivar choice are also affected by the season and the variety.

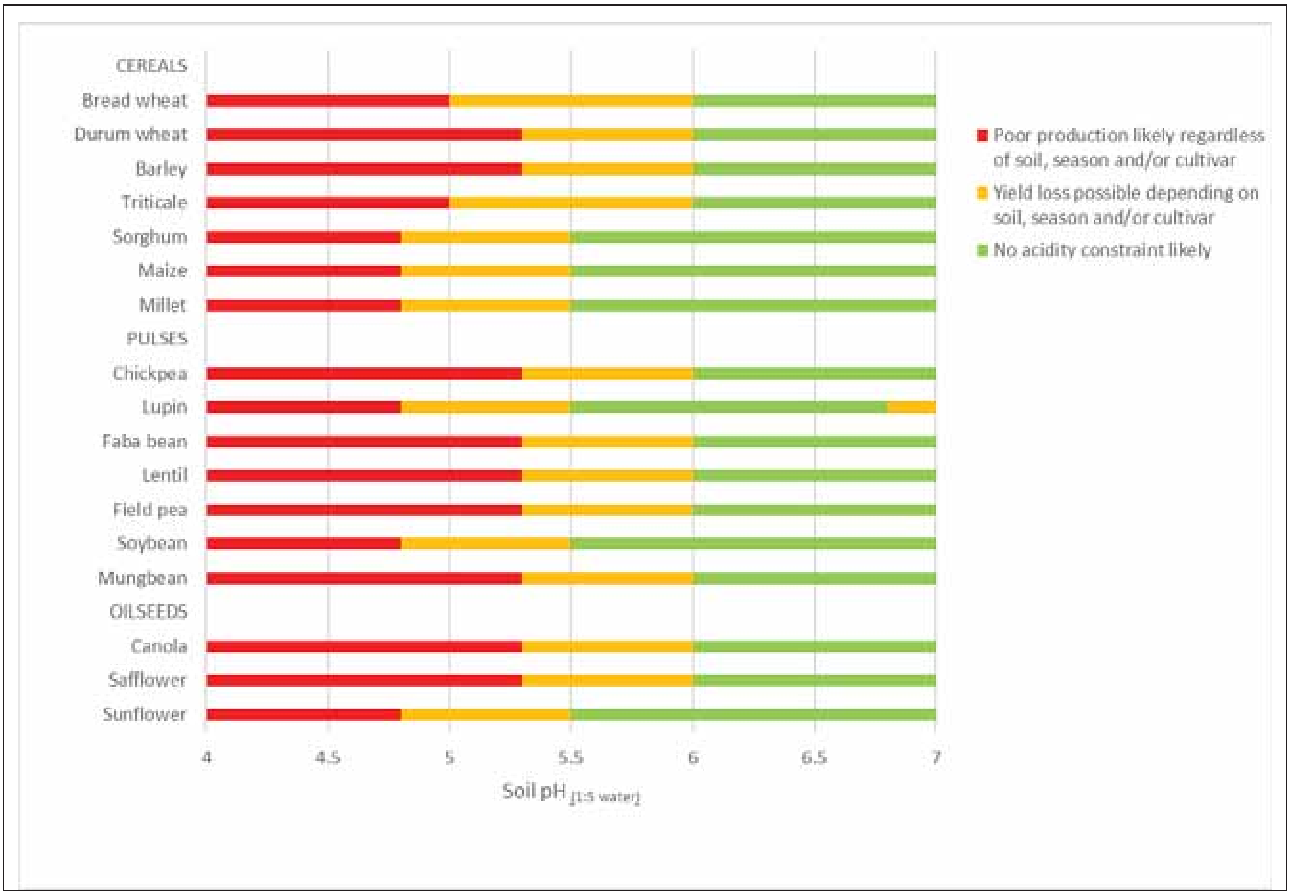


Figure 4. Soil $pH_{w1:5}$ tolerance limits proposed for crop species when grown under rainfed conditions in subtropical regions (Page et al., 2021).

Sodic soils

The literature report also indicated that tolerance to soil sodicity varies between species, although there is not currently enough information available to publish 'traffic light' boundaries for sodicity. However, it is clear that there are differences in the tolerance of different species and varieties to sodic conditions. Improving our understanding about which are the best species and varieties for areas with sodic soils is an important research priority and will help improve productivity, particularly when sodicity occurs in the subsoil and is difficult to ameliorate.

Tolerance to sodicity varies between cultivars of wheat

A limited number of cultivars have been tested at contrasting sites in several seasons

As part of a GRDC co-funded project 'Improving wheat productivity on sodic, saline and dispersive soils', (UA00159, 2015 to 2020), trials in Queensland were conducted at a highly-sodic site and a less-sodic site in each season to compare tolerance to soil constraints between certain wheat genotypes (Figure 5).

A key aim of this project was to characterise the tolerance to soil constraints of wheat genotypes in all the major Australian cropping regions. To achieve this, a core set of lines from all over Australia were tested in all regions. A small number of lines from Qld and NSW were included in each season. A comprehensive comparison of current commercial cultivars in each region was not a focus. However, data on the small number of Qld and NSW cultivars tested do show genotypic differences.

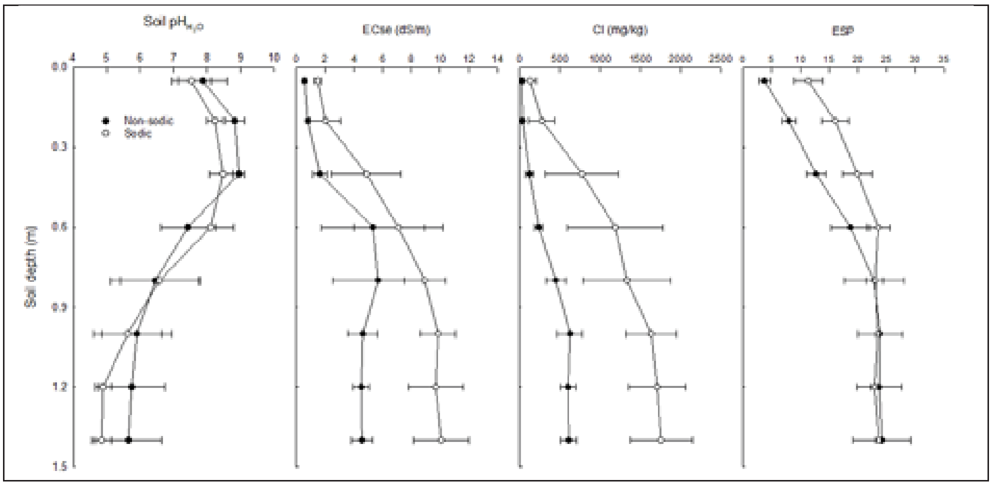


Figure 5. Average and standard errors of soil constraints for the less-sodic and highly sodic sites during 2015-19 experiments.

Soil and seasonal variability complicate field comparisons

The effects of soil constraints vary both spatially and temporally complicating attempts to compare genotypes in the field. Spatial variation in the field occurs both across the landscape and with depth in the soil profile (Figure 5). High levels of variation at the surface over distances of a few metres are common, particularly after levelling (Figure 6). There are also complex interactions among the various physio-chemical constraints (Nuttall et al., 2003). These complicated interactions can limit the agronomic and management options. Variability in the impact on the crop growth and yield is compounded by the complex interactions between the physiochemical constraints and environmental factors, particularly the timing and amount of rainfall relative to the crop development cycle. This leads to variation in genotype ranking from season to season and the ability to ascribe differences between genotypes to soil constraints.

Spatial variability

To find genetic differences in the presence of high spatial variability, it is necessary to have many plots for each genotype spread across the trial. During UA00159, 4 replicate plots per genotype were used in most years with 6 replicates in 2020. This compares to 2 or less replicates per genotype in standard multi-environment genotype trials. The need for such high numbers of replicates limits the number of lines that can be tested each year.

Figure 6. Drone image of a highly constrained trial site near Goondiwindi in 2020 showing spatial differences in canopy cover likely due to small scale differences in soil constraints.

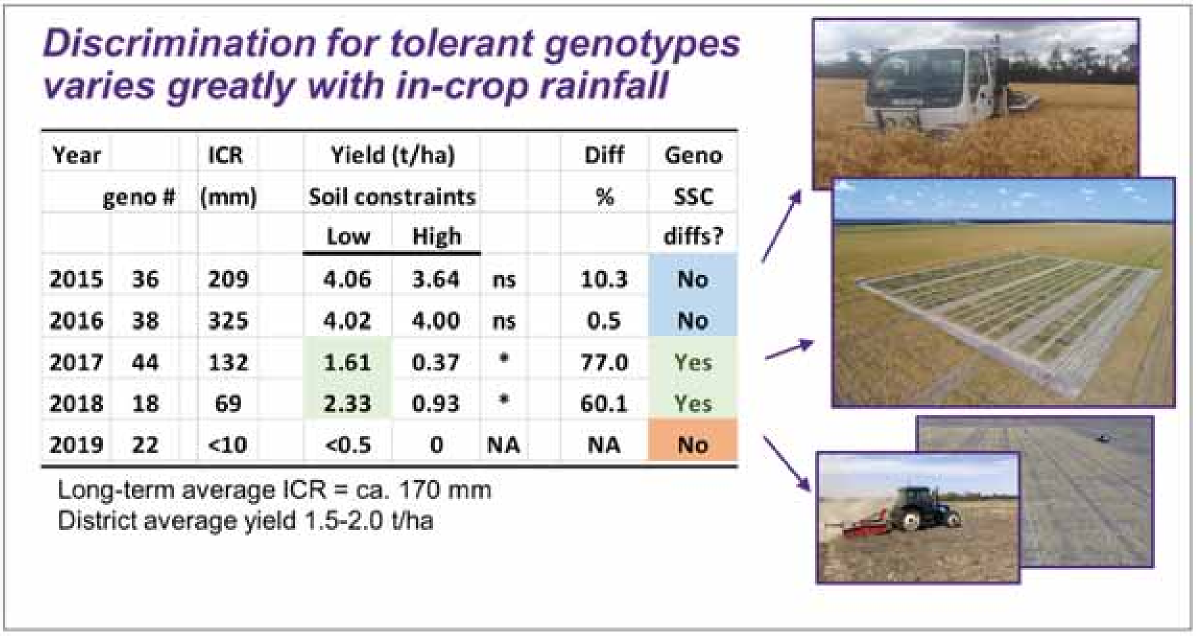
We are currently using remote sensing information from drones to determine whether information on canopy cover might be used to compensate for some of the spatial variation in soils to improve detection of genetic difference.

Temporal variability

The ability to discriminate between genotypes for tolerance to sodic soils with variable subsoil constraints in field experiments varied greatly in each season during project UA00159 (Figure 7). In 2015 and 2016, higher than average in-crop rainfall meant that plants were less reliant on access to water from the soil. Thus, rankings in these seasons are less likely to be representative of tolerance to soil constraints.

Conversely, during 2019 when in-crop rainfall was extremely low, the high-constraints trial could not be harvested at all. In dry years more generally, genotypic differences are usually much harder to differentiate because of very low yield.

In years when in-crop rainfall is near the district average, as in 2017, 2018 and 2020, discrimination between genotypes was much better.



Year	geno #	ICR (mm)	Yield (t/ha) Soil constraints Low	High		Diff %	Geno SSC diffs?
2015	36	209	4.06	3.64	ns	10.3	No
2016	38	325	4.02	4.00	ns	0.5	No
2017	44	132	1.61	0.37	*	77.0	Yes
2018	18	69	2.33	0.93	*	60.1	Yes
2019	22	<10	<0.5	0	NA	NA	No

Figure 7. Differences between mean yield at a highly sodic site and less sodic site in each season as

well as differences in genotype ranking between sites were most evident in years where in-crop rainfall (ICR) and site mean yields were close to the district average.

Genetic differences in tolerance can be found

Despite the challenges discussed above, genetic differences between varieties could be identified particularly in seasons 2017, 2018 and 2020. In general, the yield ranking of genotypes at the less-sodic site was not a good indicator of the ranking at the highly sodic site. This indicates that testing on highly sodic soils is likely to be required to identify varieties best for those sites. For example, in 2018 the cultivars Corack⊕, Mitch⊕, Trojan⊕ and Mace⊕ ranked highly at the non-sodic site, while the wheat genotypes Flanker⊕, Corack⊕, Janz and Sunco ranked highly at the sodic site (Figure 8). However, given the small number of varieties tested to date, it is not yet possible to determine sufficient economic value that would provide useful grower advice on variety choice at this time.

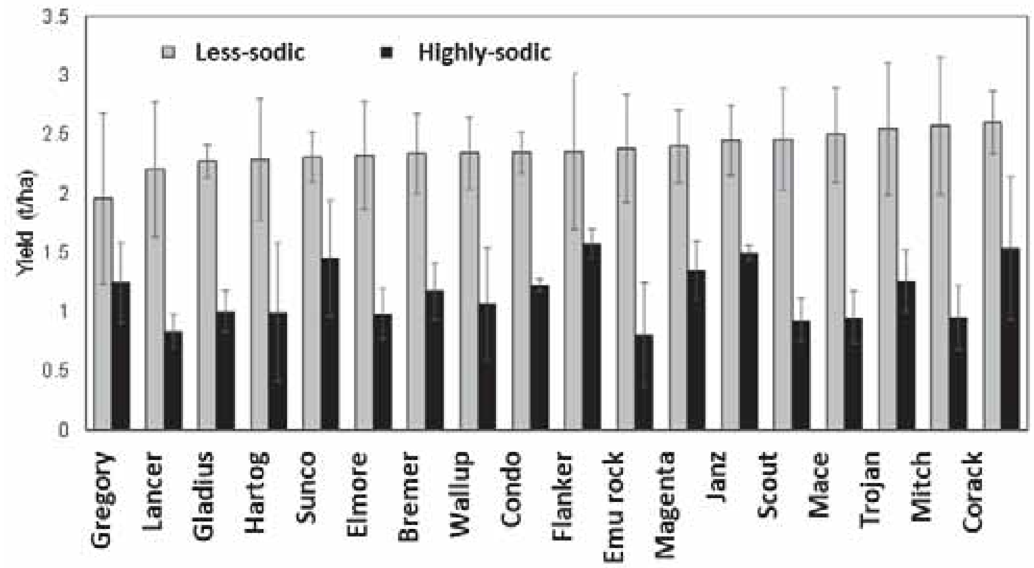


Figure 8. Mean grain yields of wheat lines at the sodic site in 2018, ranked in ascending order of yield at less-sodic site in 2018.
(⊕ The varieties Gregory, Lancer, Gladius, Elmore, Bremer, Wallup, Condo, Flanker, Emu rock, Magenta, Mace, Trojan, Mitch and Corack are protected under the Plant Breeders Rights Act 1994.)

Proxy traits could help identify prospective cultivars and breeding lines

Proxy traits are often used in research and breeding where the primary trait, in this case performance on sodic sites, is hard or expensive to measure. Field validation is always needed; however, proxy traits can sometimes be used to quickly identify genotypes most likely to perform well. Even if the proxy trait is not highly accurate in identifying the best performing lines, it may be useful to identify the vast bulk of lines that are not likely to be well adapted. This can reduce the number of lines requiring highly replicated field trials at multiple sites in multiple years.

Thus, proxy traits can potentially be used

1. To provide recommendations to growers, by helping identify the most prospective cultivars for expensive field testing
2. In breeding programs aiming to develop more tolerant lines, to screen out the bulk of low ranking lines.

Potential proxy traits have been identified

Potential proxy traits associated with tolerance to sodic soils with high Cl at depth include lower concentrations of Cl in the youngest mature leaf blades close to flowering as well as higher concentrations of calcium and potassium (Figure 8; Dang et al 2016).

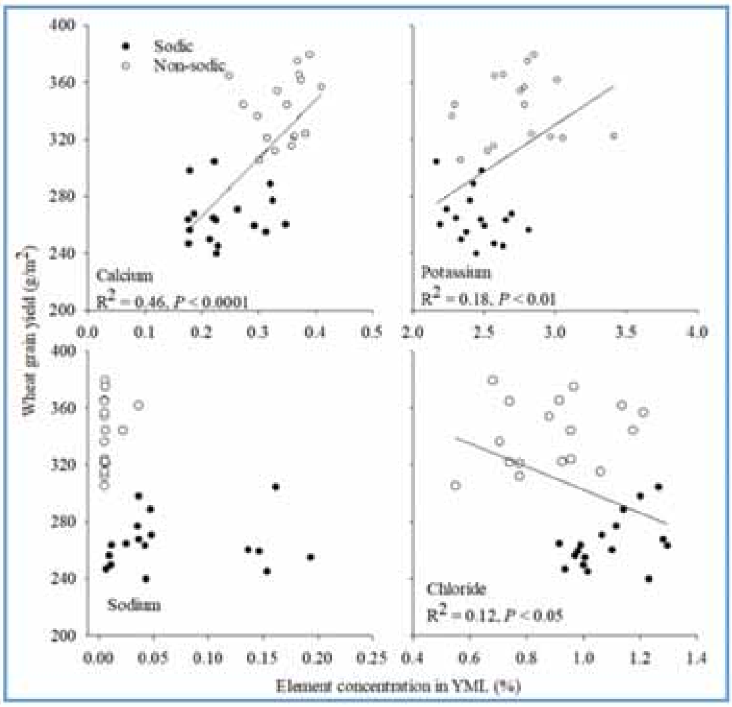


Figure 9. Concentrations of elements in the youngest mature leaf (TML) of wheat near flowering (Dang et al. 2016). Solid dots represent data for a highly sodic site and open dots a less-sodic site.

Additionally, a number of traits have been identified as associated with better establishment in the presence of a soil crust (which can often form on sodic topsoils). These include rapid germination, increased emergence force with greater hypocotyl diameter and narrower root angle (Anzooman et al. 2018, 2019).

Thus, choice of species and variety are important for optimising return on sites with soil constraints. The most tolerant species and variety differs depending on the most limiting constraint.

References

Anzooman M, Christopher J, Dang YP, Taylor T, Menzies NW and Kopittke PM (2019). Chemical and physical influence of sodic soils on the coleoptile length and root growth angle of wheat genotypes, Annals of Botany, 124(6):1043-1052

Anzooman M, Dang YP, Christopher J, Mumford MH, Menzies NW and Kopittke PM (2018). Greater emergence force and hypocotyl cross sectional area may improve wheat seedling emergence in sodic conditions Plant Science 277:188-195

Dang YP, Christopher J and Dalal RC (2016). Genetic diversity in barley and wheat for tolerance to soil constraints, Agronomy Journal, 6(4):55

Dang Y, Dalal RC, Routley R, Schwenke GD and Daniel I (2006). Subsoil constraints to grain production in the cropping soils of the north eastern region of Australia: an overview, *Australian Journal of Experimental Agriculture*, vol. 46, no. 1, pp. 19-35.

Dang YP, Routley R, Mcdonald M, Dalal RC, Singh DK, Orange D and Mann M (1999). Subsoil constraints in Vertosols: crop water use, nutrient concentrations and grain yields of bread wheat, barley, chickpea and canola, Australian Journal of Agricultural Research, 57(9): 983-98

Nuttall JG, Armstrong R and Connor D (2003). Evaluating physicochemical constraints of Calcarosols on wheat yield in the Victorian southern Mallee, *Australian Journal of Agricultural Research*, vol. 54, no. 5, pp. 487-97.

Orton TG, Mallawaarachchi T, Pringle MJ, Menzies NW, Dalal RC, Kopittke PM, Searle R, Hochman Z and Dang YP (2018). Quantifying the economic impact of soil constraints on Australian agriculture: A case-study of wheat, Land Degradation & Development, 29(11): 3866-75

Page KL, Dang YP, Dalal RC, Kopittke PM, and Menzies NW (2020). The impact, identification, and management of dispersive soils in rainfed cropping systems. *European Journal of Soil Science* 1-20 doi:https://doi.org/10.1111/ejss.13070

Page KL, Dang YP, Martinez C, Dalal RC, Wehr JB, Kopittke PM, Orton TG and Menzies NW (2021) Review of crop-specific tolerance limits to acidity, salinity and sodicity for seventeen cereal, pulse, and oilseed crops common to rainfed subtropical cropping systems, Land Degradation and Development *(under review)*

Acknowledgements

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.

The authors wish to acknowledge the support of the University of Queensland, School of Agriculture and Food Science (UQ-SAFS) also the University of Queensland Alliance for Agriculture and Food Innovation (UQ QAAFI) an alliance between the University of Queensland and the Department of Agriculture and Fisheries Queensland (DAF) as well as the excellent technical support of Scott Diefenbach and the DAF Leslie Research Facility, Farm Team.

Contact details

Dr Jack Christopher
UQ QAAFI, Centre for Crop Science.
Leslie Research Facility, PO Box 2282 (13 Holberton Street), Toowoomba
Ph: 07 4529 1314
Email: j.christopher@uq.edu.au

® Registered trademark

⦶Varieties displaying this symbol beside them are protected under the Plant Breeders Rights Act 1994.

Optimising chickpea sowing and flowering dates for maximum yield

Allan Peake[1], Elizabeth Meier[1], Kerry Bell[2], Jeremy Whish[1], M. Fernanda Dreccer[1], Tony Swan[1], Doug Sands[2], Peter Agius[2], Michael Moodie[3], Matt Gardner[4]

[1] CSIRO Agriculture and Food
[2] Queensland Department of Agriculture and Forestry
[3] Frontier Farming Systems, Mildura
[4] AMPS Research, Tamworth.

Key words

sowing date, pulse crops, chickpea, flowering date, frost risk, APSIM, agronomy

GRDC code

CSP1904-005RXT: The adaptation of pulse (chickpea and lentil) across the northern grains region.

Take home messages

- Water-limited potential yield of chickpea is constrained by frost risk, heat risk and the effect of chilling temperatures (<15°C average daily temperature) on flower abortion, so it is important to match the flowering and pod initiation phases with optimal climatic conditions
- Grain yield of chickpea can decrease by more than 250 kg/ha per week as flowering is delayed into warmer weather conditions
- The highest grain yields from field trials in 2019 were achieved with early sown crops (often with a lower harvest index than later sown crops), using the best locally adapted cultivars
- Preliminary sowing date recommendations have been prepared for a range of northern region locations for desi and kabuli chickpeas
- As 2019 was a dry season with low disease pressure, we recommend further research to test the adaptation of different cultivars to early sowing across a range of seasons.

Introduction

Chickpeas can adapt to a range of seasonal conditions, partly due to their indeterminate growth habit that allows them to continue producing branches and flowers until the crop dies due to water or heat stress. This allows the crop to replace flowers lost to environmental stresses such as frost or cool temperature flower abortion, as long as enough soil water is still available. However, there are considerable knowledge gaps in the understanding of the physiological drivers of crop growth and development for chickpea.

The GRDC funded 'Pulse Adaptation' project, studied the flowering and yield formation response of chickpea across a range of environments during the 2019 season. The project aim was to determine when flowering needs to occur to maximise grain yield, and when crops should be sown in order to achieve these flowering dates.

What did we do?

We conducted time-of-sowing experiments in nine environments across QLD and NSW in 2019, with drought conditions experienced at each site. Our four 'detailed' sites had both irrigated and rainfed treatments across three sowing dates; Gatton, Narrabri, Condobolin, and Greenethorpe (near Young), and used just 2 or 3 cultivars for each crop type. The remaining 'regional trial' locations were conducted as rainfed experiments: Emerald, Millmerran, Gatton, Come-by-Chance (near Walgett), Caragabal (near West Wyalong), and Trentham Cliffs (just over the NSW border from Mildura). Regional trials also had three sowing dates but had 4 or 5 cultivars relevant to the local environment in each experiment. Where conditions were too dry to establish on the desired sowing date, each sowing date received the same amount of irrigation (10-20 mm) to assist with germination. Sowing soil water was low at Caragabal (40 mm), moderate at Millmerran and Mildura (90 and 110 mm) and high at Emerald and Walgett (130 and 140 mm). Pests and diseases were managed using best-practice control methods for each species in each location.

We then used the APSIM model to investigate the relationship between sowing date, flowering date (specifically, the data of the appearance of the first flower on 50% of plants) and grain yield. APSIM is useful for predicting crop performance as it can be used to study the entire agricultural production system (climate, soil, crops, management) simultaneously, and in different combinations. Historical chickpea data was used along with data from the 2019 experiments to re-build the chickpea model within APSIM NextGen (i.e. the new 'Next Generation' APSIM modelling environment).

The APSIM chickpea model was refined and tested until we were confident that the model simulations of field data were acceptable. Then the model was used to predict the date of the appearance of the first flower, podding date and water-limited potential yield for the last 20 years (2000-2019), for a range of sowing dates (every fortnight from 1st March to 1st August) at 13 locations across the northern region. All simulations assumed that stored soil water on the 1st of March was 60mm (south of Dubbo) or 90 mm (north of Dubbo). Simulated row spacing was 40 cm, sowing depth was 5 cm and plant population was 30 plants/m^2. Water-limited potential yield is defined as the APSIM simulated yield of a crop for a given soil type using the rainfall and soil water available, but assuming that frost, diseases, pests and nutritional disorders have not affected yield. Studies are currently being supported by GRDC into the effect of frost on chickpea pod-set and grain yield, and these will be used to improve APSIM with a physiological frost response function once they are completed. In the interim, we used climate data and existing knowledge of frost thresholds to compare frost risk across locations.

Frost and heat-stress risk assessments were conducted by examining daily minimum and maximum temperatures with the potential for frost/heat stress (below +2°C and above +30°C at the standard met-station height of 1.2 m) across the same 20 years of climate data used in the simulations. Accumulated degrees below/above these thresholds for a given time period (e.g. last week in August) was calculated, and then compared with the time periods that recorded the most severe frost conditions at Breeza and the worst heat-stress conditions at Emerald over the entire year. This created a relative risk that could be used to compare different production environments in assessing the flowering and sowing dates needed to avoid extreme temperature events.

What did we find?

1. Model construction and testing – flowering and podding

The scientific literature shows there is variation for the relationship between photoperiod (i.e. duration of daylight in each day) and time of flowering in pulse crops (e.g. Daba et al., 2016), ranging from strong photoperiod responses to photoperiod neutral. Using data from our field trials we showed that the desi cultivars had a strong relationship between thermal time from emergence to the appearance of the first flower (i.e. date of first flower appearance on 50% of plants) and photoperiod at the end of the photoperiod insensitive phase (400 to 600 degree-days after emergence depending on cultivar). Kabuli cultivars had highly variable flowering responses ranging from similar photoperiod responses to the desi cultivars, to probable photoperiod neutral responses (data not shown). The date of first flower appearance was simulated acceptably for key desi and kabuli cultivars (Figure 1a,1b). The introduction of a new standardised phenology key for chickpea (Whish et al., 2020) will improve the consistency of phenology data collection between researchers across Australia. Further data should be collected using the new phenology key to widen testing environments and seasons and improve our understanding of the flowering response for chickpea.

The date of pod initiation is another important crop development stage in pulse crop modelling, as it represents the point at which the crop begins to partition biomass into pods and grains. A function was developed to account for chilling sensitivity of chickpea that prevents pollination and causes flower abortion at mean daily temperatures below 15°C (Clarke and Siddique, 2004). This approximated data from Srinivasan et al. (1998), Clarke and Siddique (2004) and Warren et al. (2019)

to predict the date of beginning of pod development in chickpeas as a set number of days since the appearance of the first flower when average temperature exceeded 15 degrees (default = 5). The function predicts the date that 50% of plants have at least one 1cm long pod on the main stem (when pod development is easily detectable with field scoring), so it is not predicting the date of successful pollination per se. The function worked satisfactorily in both Desi and Kabuli chickpeas (Figure 1c, 1d) but was more accurate in Kabuli chickpeas when simulating data from 2019. The desi cultivars PBA Slasher, PBA Striker and PBA Seamer all produced outliers where pod development began sooner than predicted by APSIM on the first time of sowing in certain environments. These data may be caused by the tendency for crops to begin setting pods in a spell of warm weather, after which they can be aborted when cold conditions return. Further study will be required to fully assess the ability of different cultivars to produce and retain pods under cool temperatures.

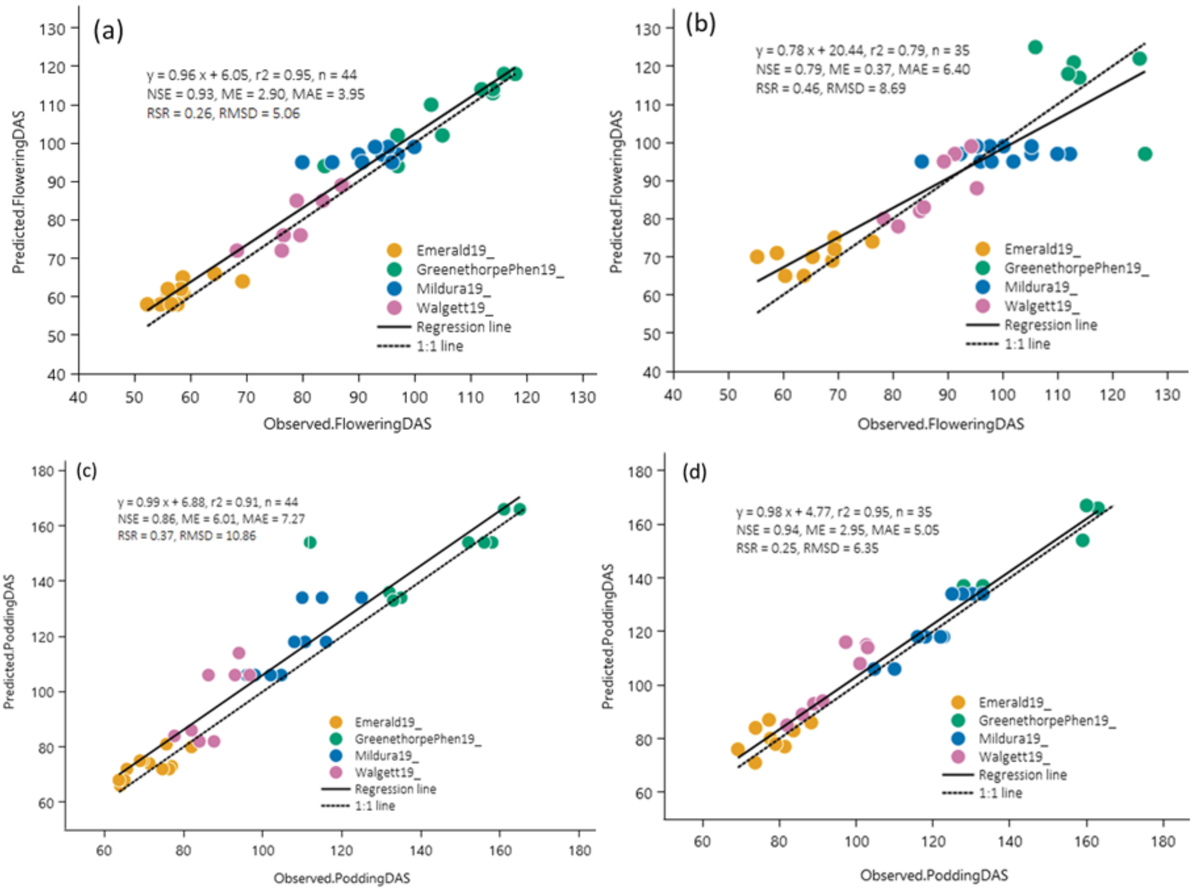


Figure 1. APSIM predicted vs observed appearance of first flower for (a) desi and (b) kabuli chickpeas, and podding date for (c) desi and (d) kabuli chickpeas, compared across regional trials in 2019.

2. Model construction and testing – grain yield prediction

The second purpose of the model was to predict water-limited potential yield, i.e. the potential yield for crops assuming that frost, nutrient deficiencies and disease are not encountered. At some point in the future it will be possible to build frost damage into the model once research into the effects of frost on chickpea is completed. In the meantime, it is helpful to understand what yields are possible if frost effects are ignored, as it shows what yield gains could be achieved by developing more frost tolerant cultivars. Model testing against experiment grain yields is presented in Figure 2.

Unlike flowering date, it isn't expected that the models will accurately simulate grain yield of every cultivar because of the local adaptation to soils and diseases that different cultivars can possess. The model is designed to simulate 'water-limited potential yield', which is represented by the best performing cultivars in each environment. While the maximum grain yield for each environment was simulated reasonably well for each crop, it was noticeable that grain yield for the earliest time of sowing was overpredicted at most locations except Emerald in the chickpea simulations (data not shown). This may have occurred because the effects of cold temperature or frost on flower viability and pod-set are not being fully simulated. Alternatively, the first time of sowing typically has the highest biomass and potential yield, so any nutritional limitations are more likely to impact grain yield in the early time of sowing. Further research is necessary to determine the reasons for these yield gaps.

An example of this is shown in Figure 3 where PBA HatTrick⍉ had similar field-measured grain yield across sowing dates and irrigation treatments at Greenethorpe, but potential (simulated) yield of the irrigated early sown treatments was much higher than measured grain yield, causing a large 'yield gap'. In this experiment PBA Slasher⍉ was much closer to achieving potential yield across sowing dates under irrigation and achieved 50% more grain yield in the irrigated trial on the first time of sowing compared to the third time of sowing, but still had a sizeable yield gap of 1 t/ha on the early sowing date. Further research is needed to determine if cultivar choice or other agronomic practices can be used to improve grain yield in conjunction with early sowing dates under favourable conditions.

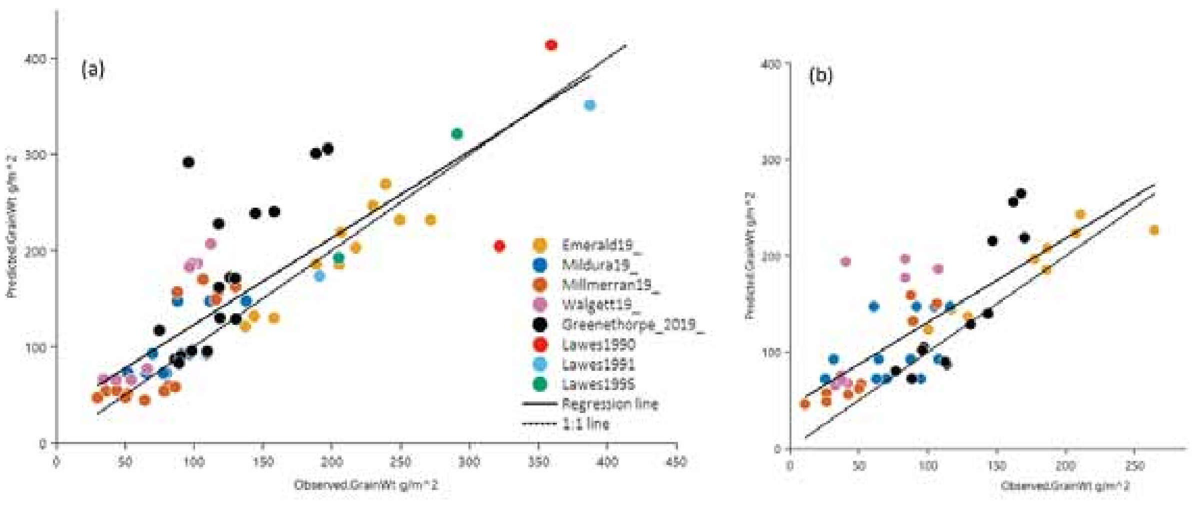


Figure 2. Predicted vs observed grain yield (a) desi and (b) kabuli cultivars, across historical data and 2019 regional trials.

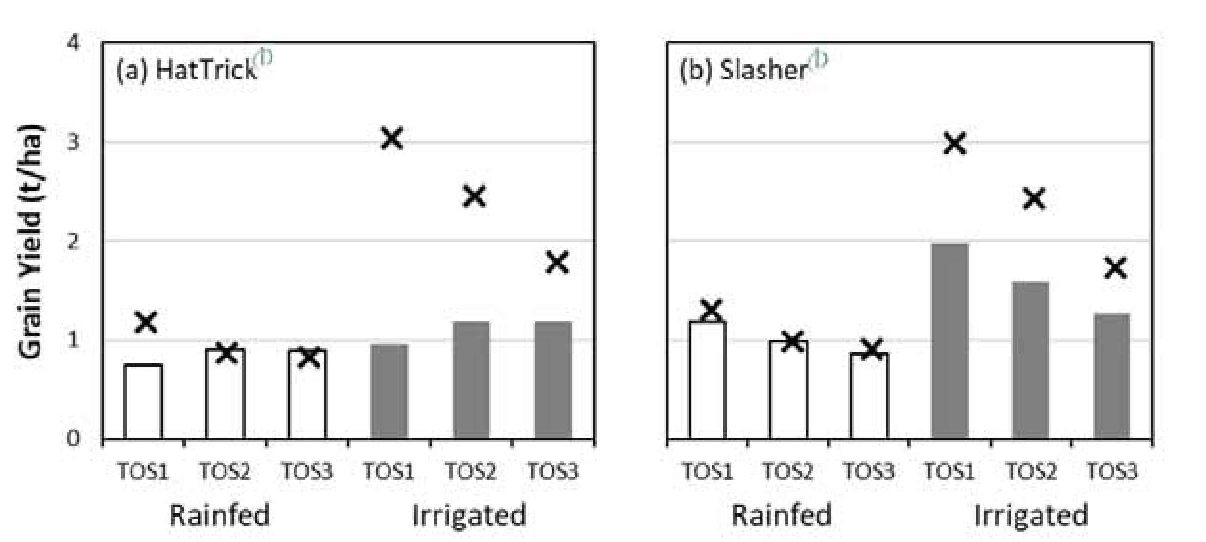


Figure 3: Measured grain yield (bars) and APSIM simulated grain yield (crosses) for the desi cultivars (a) PBA HatTrick⌽ and (b) PBA Slasher⌽ across three times of sowing (TOS) at Greenethorpe, 2019. Sowing dates were 30-April, 21-May, 12-Jun. Standard error is ≤ 0.1 t/ha for all measured yield data.

3. Regional field trial results

Despite the tendency of early sown crops to have larger yield gaps as discussed above, early sowing still achieved the best grain yield in our regional field trials in 2019 for at least some cultivars at each location (Figure 4). Our earliest sowing dates in southern NSW were not as early as the earliest sowing date of Richards et al. (2020) who observed lower grain yield in crops sown on the 15th of April compared to the 30-April in southern NSW, also in 2019. The focus of our final activity for this project was understanding the optimum flowering time to avoid frost and heat shock and maximise grain yield.

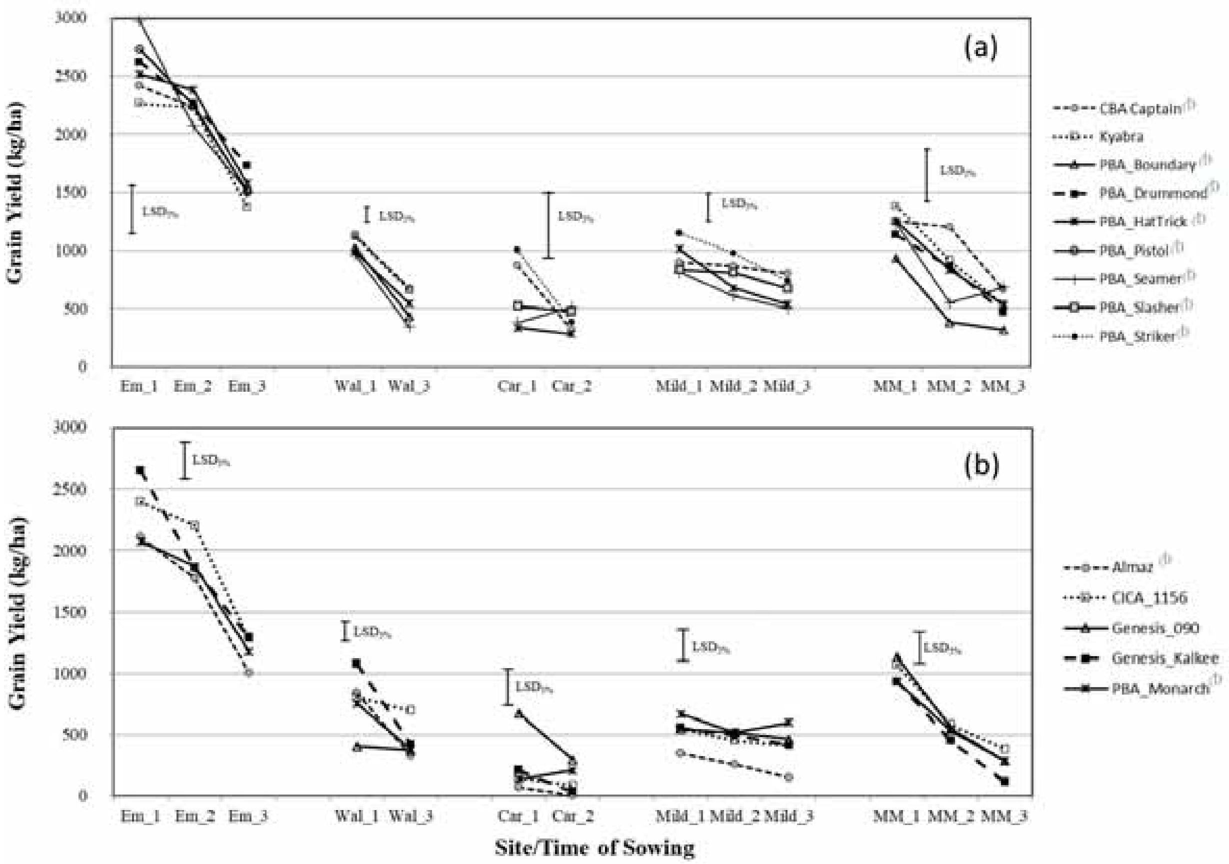


Figure 4. Grain yield vs time of sowing and location for (a) desi chickpea, and (b) kabuli chickpea from three sowing dates in five rainfed experiments in 2019. Sites and sowing dates were: Em = Emerald (10-May, 3-June, 27 June), Wal = Walgett (21-May, 28-Jun), Car = Caragabal (8-May, 17-Jun), Mild = Mildura (1-May, 18-May, 7-Jun), MM = Millmerran (17-Jun, 2-Jul, 15-Jul). Only two sowing dates were achieved at Caragabal due to low soil moisture and lack of rainfall during the sowing window. The second sowing date at Walgett for desi and kabuli chickpea were destroyed by wild pigs prior to emergence.

4. Variation of optimum flowering window, sowing date and potential yield across the northern region

Our ultimate aim was to use APSIM to show how flowering time (measured as the appearance of the first flower) varies with sowing date across the northern region. In addition, we linked this information with water-limited potential yield and an assessment of relative frost/heat risk, to help growers understand when they should be sowing chickpea to achieve maximum grain yield. The data is presented in a series of charts (Figure 5-10). Due to the number of locations we have simulated, data for each crop or subspecies is presented across two pages, with the more northern sites on the first page, and locations in central and southern NSW presented on the second page. Graphs for Emerald and Mildura are presented on a separate page as they have different optimum sowing periods to most other locations, with Dalby and Meandarra on the final page of graphs as a late addition for these updates in 2021.

For ease of presentation we have presented results for one cultivar for each chickpea type (i.e. PBA HatTrick⍉ for desi chickpeas and Almaz⍉ for kabuli chickpeas). If quicker/slower maturing cultivars are chosen by growers then sowing date should be adjusted to be slightly later or earlier. Local knowledge can be used in the interim to decide whether alternative cultivars will flower earlier or later than our simulated cultivars. It should be noted that the flowering benchmark used in this study is the appearance of the first flower on 50% of plants, which can occur much earlier than the date a crop is flowering prolifically and is not always easy to identify in the field.

Interpreting the potential yield graphs

Potential yield is 'water-limited': i.e. a simulated or predicted yield that uses the historical rainfall and available soil water, but assumes that frost, diseases, pests and nutritional disorders have not affected yield. As discussed above, the model does account for the effect of cool temperatures on flower pollination/abortion (e.g. Clarke and Siddique, 2004) by delaying the beginning of pod-set, so the emphasis herein is on the risk of intermittent frost events that may cause yield-limiting damage to floral structures and developing pods. In the following figures, the box and whisker plots have a small black line which indicates the median yield. The yield from 50% of years fall inside the box (i.e. yields from 10 years, with five years either side of the median yield), and the box whiskers represent 1.5 times the value of the adjacent quartile (i.e. between the median and the edge of the box), while grey dots represent outliers beyond this range.

Determining optimal sowing date

In the following figures, a shaded box is used to connect three-graph sets from the same location showing the predicted optimal time of flowering and pod initiation. The beginning of this 'optimum flowering window' is determined by assuming that the earliest flower needs to appear in the last fortnight when relative frost risk is still above 20% (i.e. approximately one day with a minimum temperature of 0°C in the week), as additional experiments suggest that the most sensitive growth stage to stress possibly falls 100 degree days later than the appearance of the first flower (Dreccer et al., 2020). The end of the optimum flowering window is the point when relative heat risk of 20% is avoided, or when 5 weeks have elapsed since the window opened. An arrow has been inserted in the week where average temperature increases above 15 degrees (and the risk of cool temperature flower abortion is reduced), to help demonstrate when pod-set is likely to begin. In the middle graph of each set it is possible to determine which sowing dates can be used to achieve the optimal flowering dates, by referring across to the y-axis. Potential yield data to the left of the optimum flowering window are at high risk of not being achieved due to frost risk.

Additional important notes

1. These simulations were conducted using an average sowing depth of 5cm. The effect of sowing depth on chickpea phenology may vary across the northern region, and further study is needed to determine how much earlier deep-sown crops should be sown to achieve the flowering dates presented here.
2. Frost risk varies markedly within short distances. Data in these figures may not apply to farms that are closely situated to the locations for which we have provided simulations, due to differences in elevation and aspect between met stations and individual fields.
3. Simulations have been prepared using 60-90 mm of stored soil water prior to sowing, depending on location (i.e. 60 mm south of Dubbo, 90 mm north of Dubbo). Water-limited potential yield will vary from what we have presented, if more or less water is available at sowing.

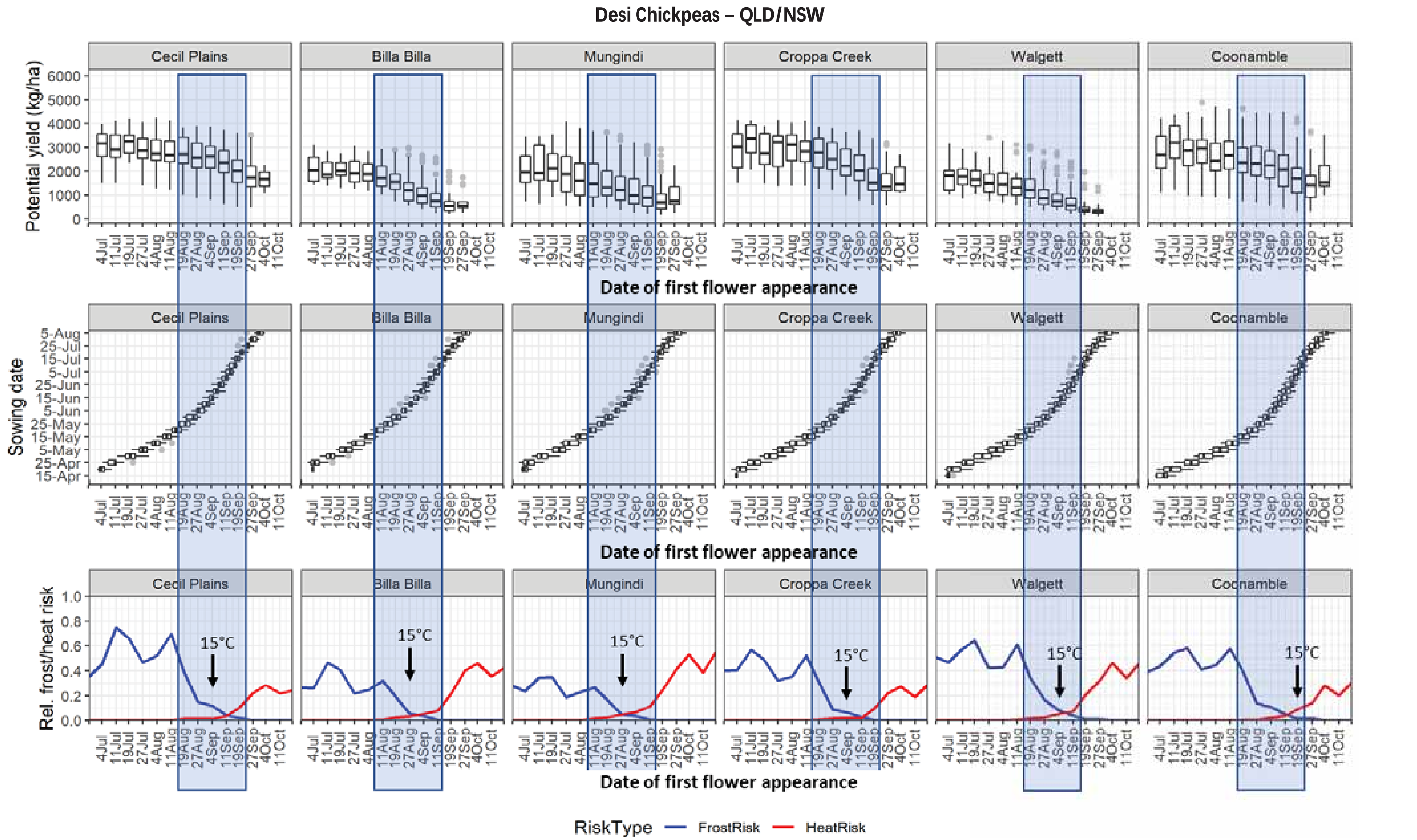


Figure 5. Water-limited potential yield, sowing date and relative frost/heat risk, plotted against the date of first flower appearance for PBA HatTrick⦶ at six environments in QLD and northern NSW. Shaded area represents the predicted optimum window for appearance of first flowers. Arrows and '15°C' mark the first week that has an average daily temperature ≥15°C in the second half of the year, i.e. when conditions become favourable for pod-set.

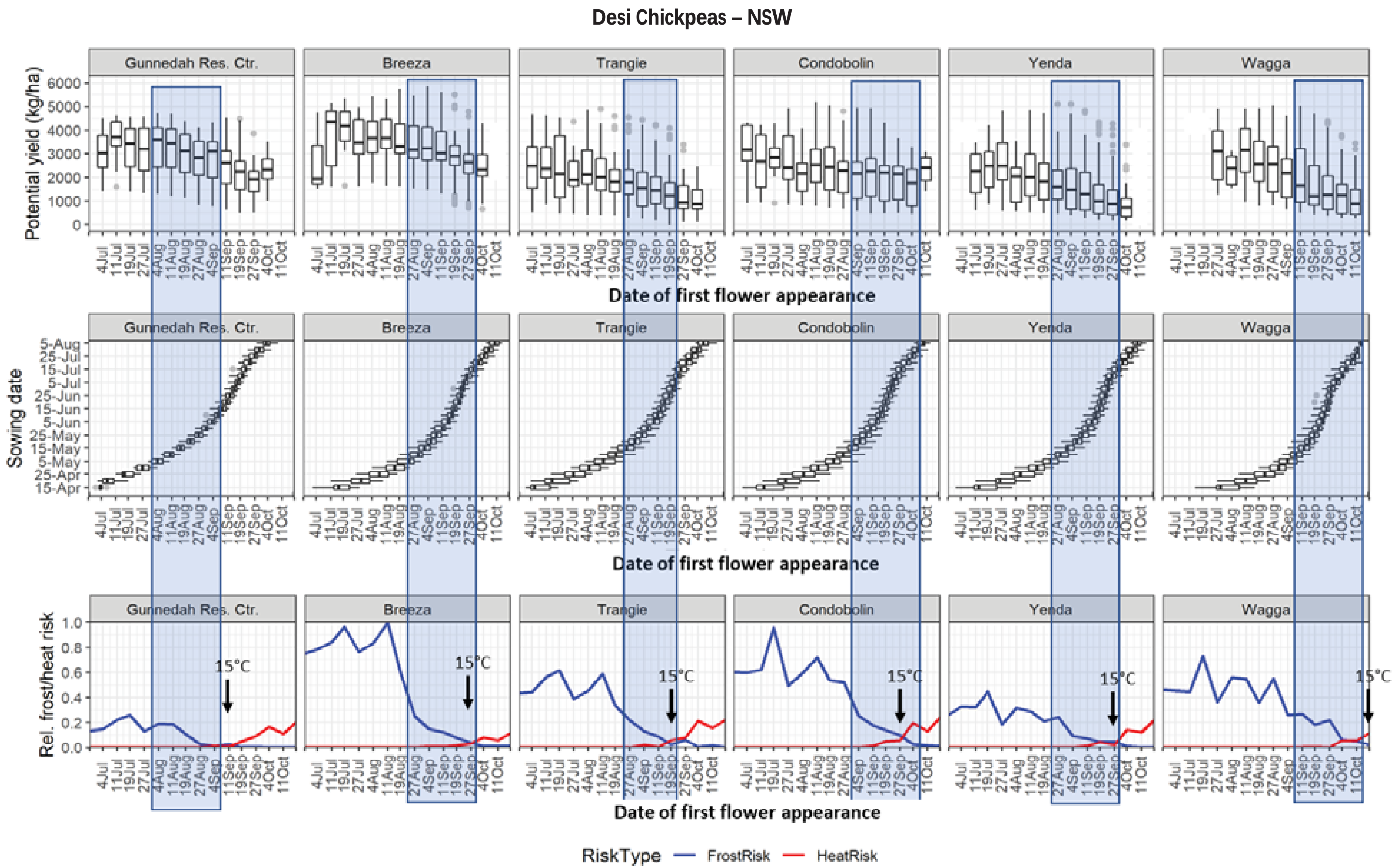


Figure 6. Water-limited potential yield, sowing date and relative frost/heat risk, plotted against the date of first flower appearance for PBA HatTrick at six environments in NSW. Shaded area represents the predicted optimum window for appearance of first flowers. Arrows and '15°C' mark the first week that has an average daily temperature ≥15°C in the second half of the year, i.e. when conditions become favourable for pod-set.

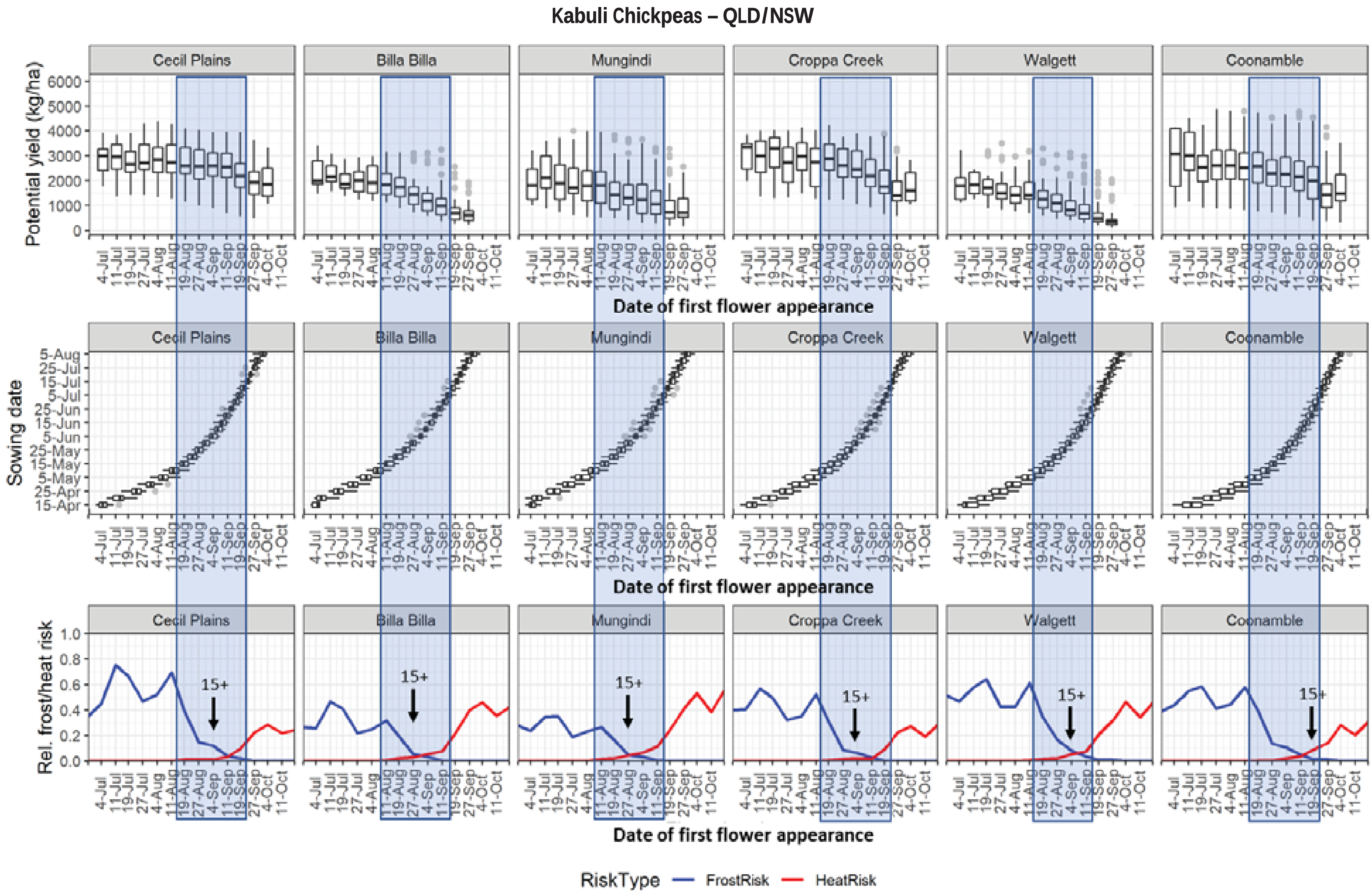


Figure 7. Water-limited potential yield, sowing date and relative frost/heat risk, plotted against the date of first flower appearance for Almaz⏀ at six environments in QLD and northern NSW. Shaded area represents the predicted optimum window for appearance of first flowers. Arrows and '15°C' mark the first week that has an average daily temperature ≥15°C in the second half of the year, i.e. when conditions become favourable for pod-set.

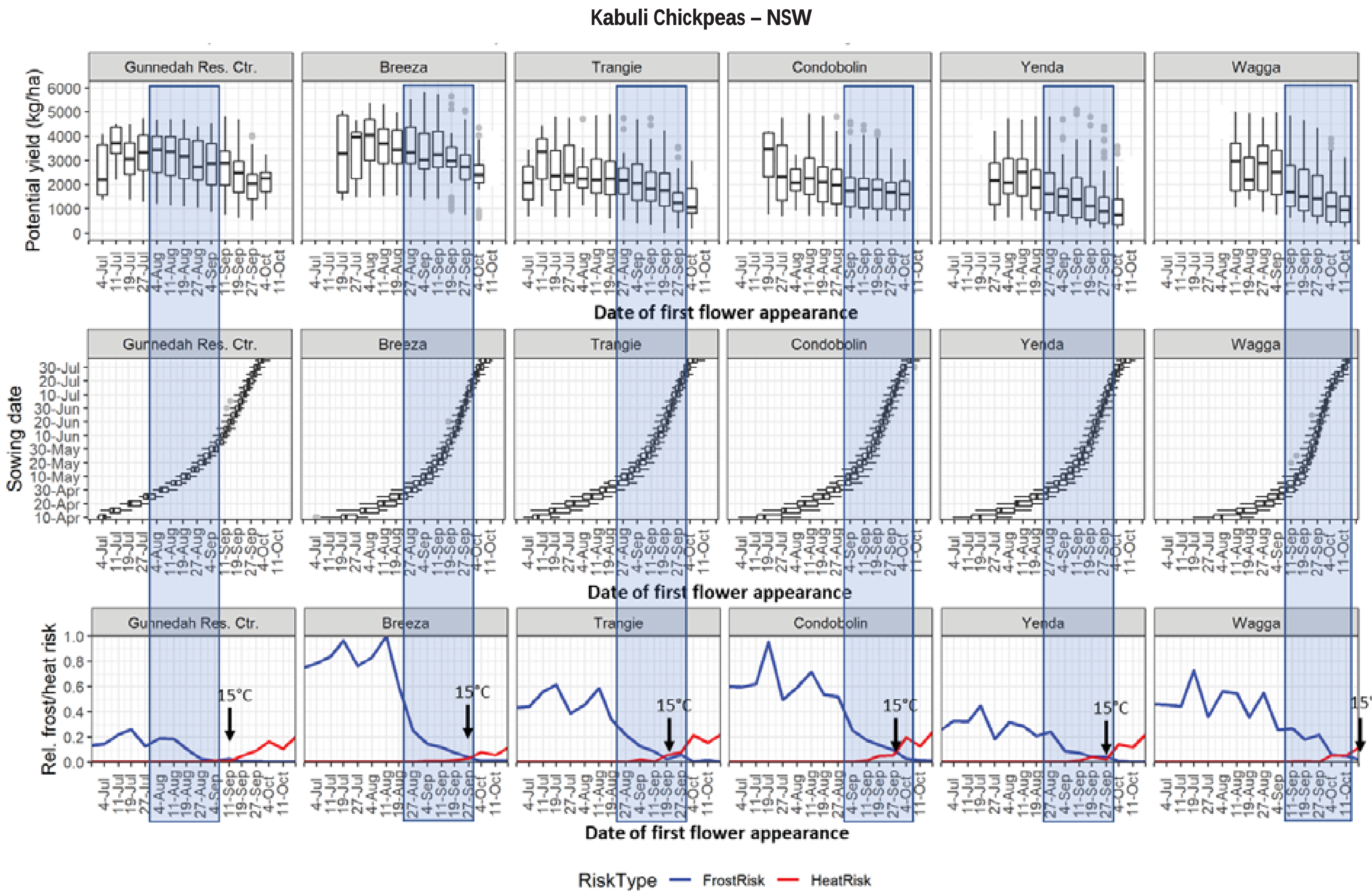


Figure 8. Water-limited potential yield, sowing date and relative frost/heat risk, plotted against the date of first flower appearance for Almaz⊕ at six environments in NSW. Shaded area represents the predicted optimum window for appearance of first flowers. Arrows and '15°C' mark the first week that has an average daily temperature ≥15°C in the second half of the year, i.e. when conditions become favourable for pod-set.

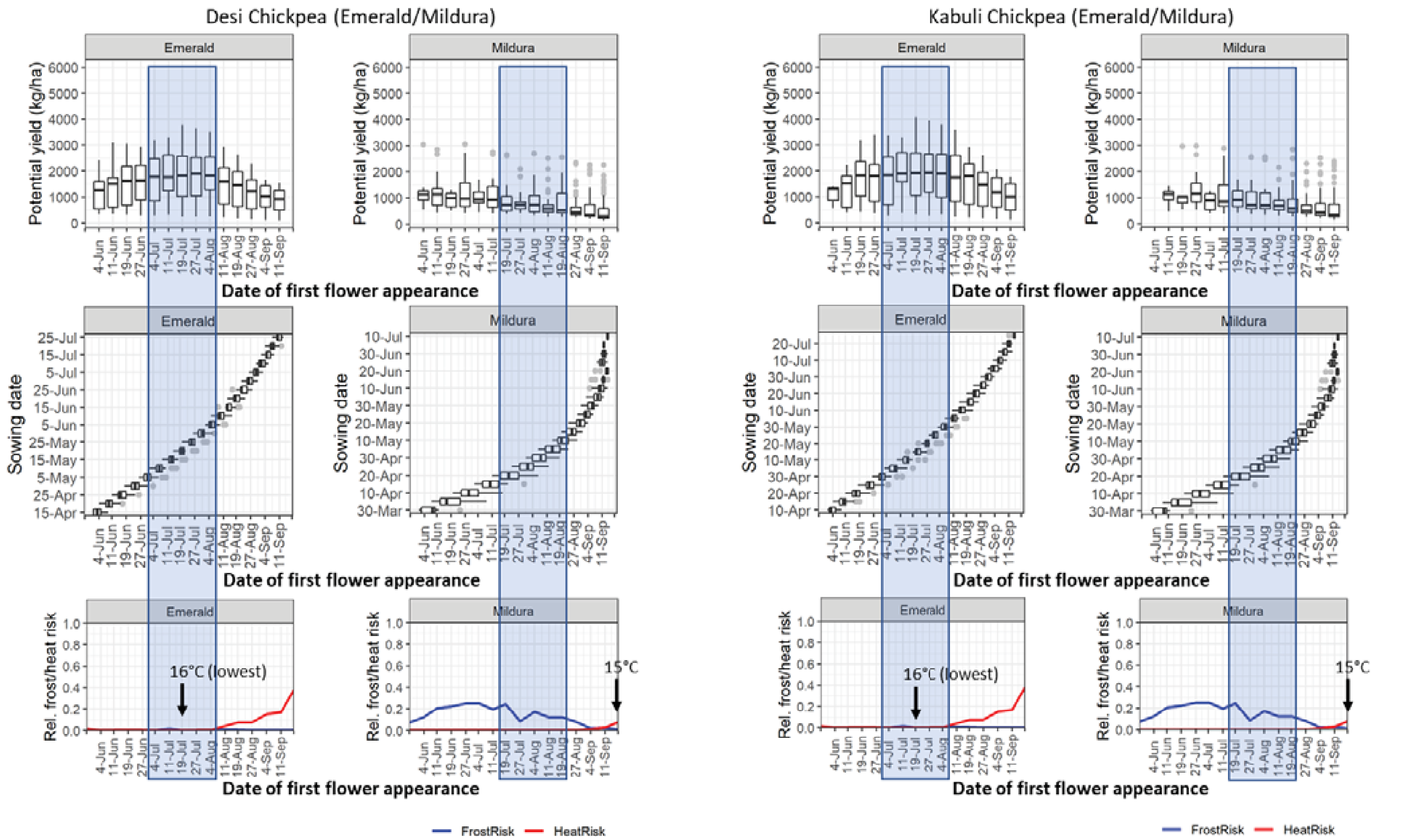


Figure 9. Water-limited potential yield, sowing date and relative frost/heat risk, plotted against the date of first flower appearance for PBA HatTrick⦶ and Almaz⦶ at Emerald and Mildura. Shaded area represents the predicted optimum window for appearance of first flowers. Arrows and '15°C' mark the first week that has an average daily temperature ≥15°C in the second half of the year, i.e. when conditions become favourable for pod-set.

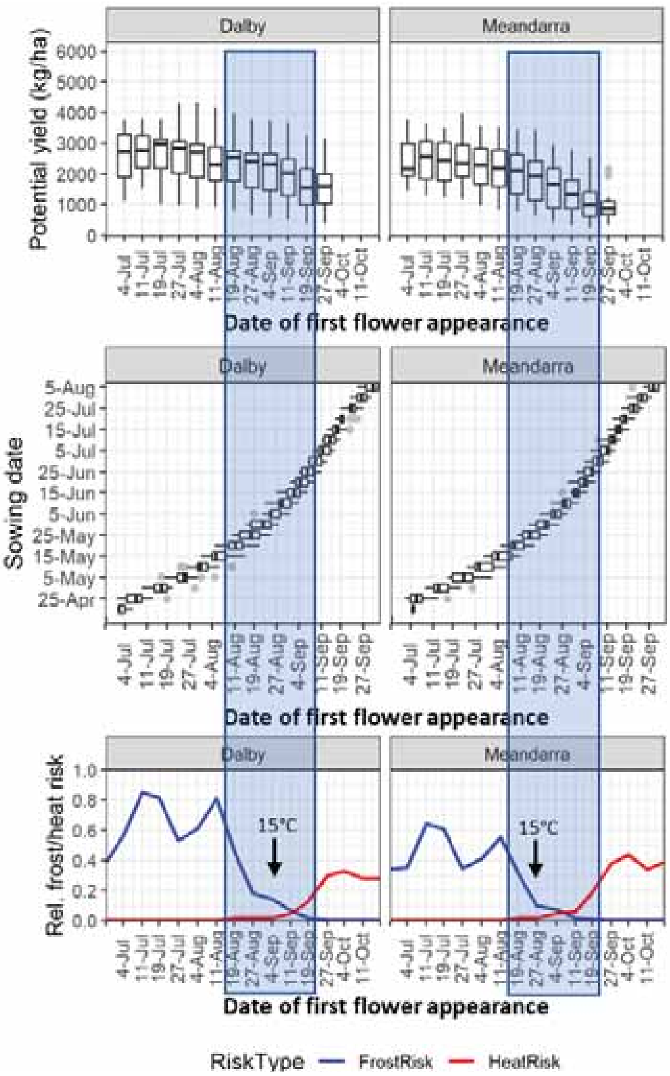


Figure 10. Water-limited potential yield, sowing date and relative frost/heat risk, plotted against the date of first flower appearance for PBA HatTrick⚘ at Dalby and Meandarra. Shaded area represents the predicted optimum window for appearance of first flowers. Arrows and '15°C' mark the first week that has an average daily temperature ≥15°C in the second half of the year, i.e. when conditions become favourable for pod-set.

Trends and observations

Water-limited potential yield declines rapidly in spring with later sown crops, particularly in the more northern environments.

Within the optimum flowering window, yield declined up to 300 kg/ha per week that first flower appearance was delayed in the most extreme situations (e.g. Cecil Plains, Billa Billa (Figure 5) and Meandarra (Figure 10)). Although the model showed a tendency to over-predict grain yield on early sowing dates in some environments (Figure 3), yield was lower for later sowing dates in the field trials (Figure 11), where grain yield declined at between 60 and 225 kg/ha per week that the first flower appearance was delayed, depending on location and time-of-sowing. While it can be difficult to determine if yield reductions are due to heat stress, water stress or simply the shorter growing season experienced by late sown crops, delayed flowering into warmer weather conditions reduced yield in the 2019 field trials and the simulations. However in wet seasons delayed sowing may not have the same yield penalty, so further research is necessary to investigate the relationship between time of sowing and yield across a range of locations and seasons.

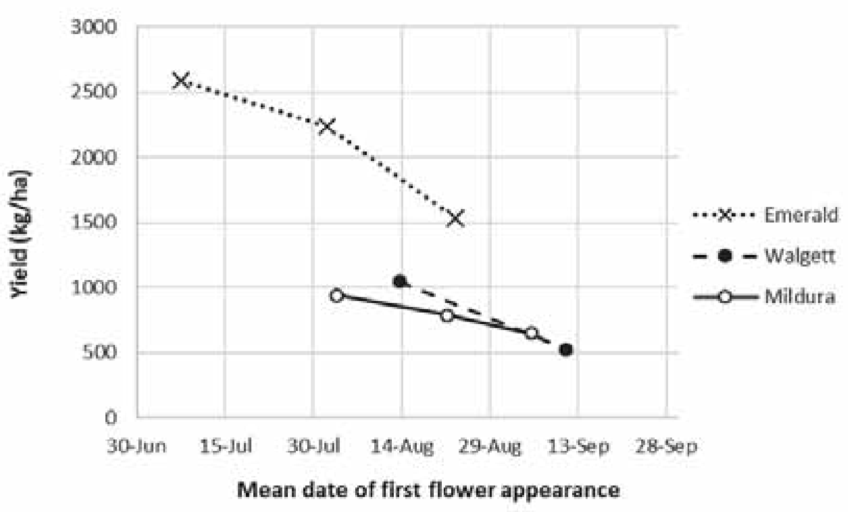


Figure 11. Mean grain yield vs mean date of first flower appearance across 5 desi chickpea varieties from three 'time-of-sowing' field experiments conducted in 2019. Markers (cross or circles) represent data generated from a single sowing date. The Caragabal and Millmerran experiments were not included due to uneven emergence that prevented accurate assessment of plot flower appearance.

It is unclear whether ultra-early sowing could provide even greater yield advantages in a dry season.

While most northern sites reached the 15°C threshold for pod-set 2-3 weeks after the beginning of the optimum flowering window, the more southern sites had a larger gap (e.g. 5 weeks at Wagga and 9 weeks at Mildura). While early sown crops may not set pods for many weeks after the first flower appears, the increased biomass development and/or root growth that occurs during this delay may be important for maximising grain yield in a dry season. While Richards et al. (2020) identified a yield penalty associated with ultra-early sowing in southern NSW, their results were obtained in different environments to those of southern QLD and northern NSW. Further research is recommended in both high and low frost-risk environments with a range of stored soil water to understand how long this infertile flowering phase can continue before significant yield loss occurs. This research would help define the optimum flowering window and sowing dates with more precision for a range of environments and seasons.

Frost risk varies over short distances within the landscape, so individual farms or paddocks may have different risk profiles (and optimum sowing dates) to those presented here.

Recent work by AMPs Research in wheat has shown that the top sections of some sloping paddocks on the Liverpool Plains can be sown 4-6 weeks earlier due to lower frost risk. Similarly, it is important to understand that the charts presented here may not be applicable to nearby farms or paddocks due to differences in local topography. For example, our first simulations for the Liverpool Plains used the Gunnedah Resource Centre meteorological station, but we later realised this data was not representative of most farms in the Gunnedah district. We then compared these simulations with the equivalent data from nearby Breeza (side-by-side comparison in Figure 6 or 8) showing a much greater frost risk at Breeza compared to the Resource Centre for the same dates in August.

Growers should seek independent advice on frost risk for their individual locality and be careful about making large changes to their normal sowing dates based on data presented here.

Kabuli cultivars are generally slower maturing than desi cultivars.

Growers swapping between the two types for the first time need to be mindful of sowing Kabuli chickpeas earlier than desi chickpeas, to flower in the same optimal flowering window.

Extremely early and ultra-late sown crops have the lowest water-limited potential yield.

Some growers in southern NSW are experimenting with spring-sown chickpea to avoid the overly wet, winter growing conditions they encounter in good seasons. However, they may struggle to generate acceptable grain yield unless they are in a cool environment and/or have a genuinely full profile of moisture, and experience good spring rainfall. Further simulations could be conducted to investigate these possibilities.

Effect of deep sowing and water stress on optimum sowing date

As discussed above, the predictions of optimum sowing date in Figures 5-10 assume a sowing depth of 5cm. As chickpea is often sown deeper than this, it is possible growers may need to take this into account when deep sowing, by sowing earlier than the sowing dates presented here to achieve the same flowering date. However, experiments in Western Australia on sandy soils have shown negligible differences for emergence date and flowering date for an early-season chickpea experiment sown at 5 and 20 cm (Rich and Lawes, 2020). Additional anecdotal evidence suggests that while the delay may be small in a warm environment (e.g. Central QLD), it may be larger in cooler environments such as the Liverpool Plains. A rule of thumb used by some agronomists is to allow one days delay in emergence for every additional centimetre of sowing depth, however it is possible that deep sown seedlings may be accumulating 'heat units' toward their phenological development while underground, so delayed emergence may not lead to an equivalent delay in the appearance of the first flower. More research is necessary to determine the effect of sowing depth on emergence date and phenology processes across QLD and NSW pulse production regions. Growers and agronomists are advised to keep records on this information for individual fields, as it probably varies between soil types and location.

Additional experiments from 2019 also showed that flowering date sometimes occurred earlier in rainfed plots compared to irrigated plots, suggesting that water stress may hasten reproductive processes. The trend was not consistent across locations and cultivars, but in the most extreme case (Genesis 90 at Greenethorpe) flowering processes were 10-19 days earlier in rainfed treatments compared to irrigated treatments, while at Gatton no differences were detected in flowering date for any cultivar but podding date was 4-12 days earlier in rainfed treatments of both PBA HatTrick⦶ and Genesis 90. The degree of water stress was probably different between these locations, which would explain the variable results. Further work is needed to determine how different severity of water stress might impact on the occurrence of the optimal flowering window for specific situations and soil types. Wet conditions might reasonably be expected to delay flowering (and hence require earlier sowing to achieve the same flowering date) as has been observed in crops such as wheat. However, disease control may be more difficult in conjunction with early sowing in wet seasons, so optimum sowing date from a grower's perspective may ultimately not differ greatly between wet and dry years.

Conclusions

The use of crop models in conjunction with field experiments has successfully transformed agronomic practices across Australia in the last three decades. The investment made in improving modelling capacity of chickpea through this project has increased confidence in the prediction of flowering date, potential yield, optimum flowering windows and the sowing dates needed to achieve them. We have also demonstrated the need for further research into cultivar response to early sowing, understanding the optimum length of the infertile flowering phase, and improving our understanding on the effect of water stress and sowing depth on phenology.

The process of investigating experiments using the models also provides direction for future research by highlighting the scientific 'bottlenecks' that improved prediction and understanding of crop performance. Additional research is needed across additional seasons to ensure that the predictions of crop performance apply to a range of seasonal conditions, not just the extreme drought conditions experienced during 2019. Model testing in additional environments and seasons will undoubtedly highlight further strategic research priorities that could be addressed to benefit Australian grain farmers into the future.

Acknowledgements

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, and the authors would like to thank them for their support. CSIRO, NSWDPI, SARDI, QDAF, USYD, UQ-QAAFI, The Australian Federal Government, QLD, NSW and SA State Governments are also acknowledged for directly and indirectly contributing to project funding. We would also like to thank Zvi Hochman and Heidi Horan (CSIRO), Richard McCallum, Mark Richards and Kirsty Hobson (NSWDPI), Helen Bramley and Josie Clarke (University of Sydney) for their contributions to the project that were not reported in this paper. We thank Paul McIntosh (Pulse Australia and the Australian Herbicide Resistance Initiative) for assisting with disease control protocols, and Neil Huth (CSIRO) for assisting with APSIM model development. We also acknowledge the incredible work carried out by the many technical staff from many organisations in successfully planning and conducting the extensive field trial program, which was instigated at short notice.

References

Clarke, H. J. and Siddique, K. H. M. (2004). Response of chickpea genotypes to low temperature stress during reproductive development. *Field Crops Research*, *90*, 323-334.

Daba, K., Warkentin, T. D., Bueckert, R., Todd, C. D., & Tar'an, B. (2016). Determination of photoperiod-sensitive phase in chickpea (Cicer arietinum L.). *Frontiers in Plant Science*, *7*, 478.

Dreccer, M.F., Sadras, V., Lake, L., Peake, A.S., Lee Long, I. et al. (2020). Appendix B: Understanding the critical stage for yield formation in chickpea and lentil in response to abiotic stress. *In* Peake A.S. et al. (Eds) 'Final Report to GRDC project CSP1904-005RXT: The adaptation of pulses (chickpea and lentil) across the northern grains region'. CSIRO Agriculture and Food, ACT, Australia.

Rich, S. and Lawes R. (2020) Sowing flexibility of chickpea and lentil in the Western Australian farming system. GRDC Updates, February 2020, Perth.

Richards, M., Maphosa, L., Presto, A., Napier, T., Hume, I. (2020). Pulse adaptation – optimising grain yield of chickpea and lentils. GRDC Northern Region Updates, 2019. Accessed online at: https://grdc.com.au/resources-and-publications/grdc-update-papers/tab-content/grdc-update-papers/2020/02/pulse-adaptation-optimising-grain-yield-of-chickpea-and-lentils

Srinivasan, A., Johansen, C., & Saxena, N. P. (1998). Cold tolerance during early reproductive growth of chickpea (Cicer arietinum L.): characterization of stress and genetic variation in pod set. *Field Crops Research*, *57*(2), 181-193.

Warren, A., Graham, N., Raman, R., Hobson, K. (2019). The physiology and genetics of cold temperatures in chickpeas – what do we know and where is the research heading? GRDC Northern Region Updates, 2019. Accessed online at: https://grdc.com.au/resources-and-publications/grdc-update-papers/tab-content/grdc-update-papers/2019/02/the-physiology-and-genetics-of-cold-temperatures-in-chickpeas-what-do-we-know-and-where-is-the-research-heading

Whish, J., Peake, A.S., Rodriguez, D., Swan, T., Richards, M., et al. (2020). Appendix F: Chickpea Phenological Key. *In* Peake A.S. et al. (Eds) 'Final Report to GRDC project CSP1904-005RXT: The adaptation of pulses (chickpea and lentil) across the northern grains region'. CSIRO Agriculture and Food, ACT, Australia.

Contact details

Allan Peake
CSIRO Agriculture and Food
PO Box 102, Toowoomba, 4350
Ph: 07 4571 3212
Email: allan.peake@csiro.au

⦶ Varieties displaying this symbol beside them are protected under the Plant Breeders Rights Act 1994.

The drivers of phenology for more profitable sowing decisions - balancing risk of abiotic stresses when selecting cereal varieties to optimise yield potential in southern Qld.

Felicity Harris, NSW DPI

For Felicity Harris and Rick Graham's paper on phenology presented at the Narrabri GRDC Grains Research Update, please go to page 114 of these proceedings

Notes

Fall armyworm update

Dr Melina Miles[1] and Dr Lisa Bird[2]

[1] *Queensland Department of Agriculture and Fisheries*
[2] *NSW Department of Primary Industries*

Key words

Fall armyworm, insecticide resistance, identification, crop damage

Take home messages

- Fall armyworm (FAW) is now widespread in all growing regions of the northern grains region
- Monitoring of susceptible crops including maize (for grain or silage), and sorghum is now essential at crop establishment and vegetative stages. Monitoring from tasselling and head emergence in these crops should now include FAW
- Natural enemies are very active on FAW and it is important to look for them when checking crops
- Recent testing has revealed established insecticide resistance in Australian FAW populations. A planned approach to insecticide selection and rotation is required to minimise further resistance development and optimise the cost-effectiveness of insecticide applications.

Fall armyworm resistance screening research

Reliance on chemical control in the management of FAW on a global scale over many decades has led to the development of resistance to at least 29 insecticide active ingredients in six mode-of-action groups.

To understand how this will impact the effectiveness of chemical control options, NSW DPI with support from the CRDC, has conducted research to establish the toxicity profiles of insecticide groups currently registered under permit for FAW in Australia (Table 1). This information will assist growers in deciding the most appropriate course of action for managing outbreaks of FAW if sprays are warranted.

The screening procedures were adapted from those previously developed for laboratory-based resistance testing in Helicoverpa. In the case of broad-spectrum contact insecticides (such as synthetic pyrethroids and carbamates), testing was done by topical application of insecticide as a backline treatment to FAW larvae. On the other hand, selective insecticides (such as the Group 28s, indoxacarb, spinosyns and avermectins) are more active by ingestion than by contact. Therefore, these insecticides were administered by adding formulated insecticide to an artificial diet on which the insect larvae were fed. Since all insecticides are also registered for *Helicoverpa armigera* (cotton bollworm), the dose response in *H. armigera* was used as a standard for comparing the relative efficacy of products in FAW.

The key findings from the research are;

- A similar level of toxicity (efficacy) of emamectin benzoate (e.g. Affirm®) and Group 5 insecticides spinosad and spinetoram (e.g. Entrust® and Success Neo®) in both *H. armigera* and FAW at all levels of the dose response.

- A similar level of toxicity (efficacy) of the Group 28 insecticide chlorantraniliprole (e.g. Altacor®) at high doses in *H. armigera* and FAW. However, FAW was about 2 times less sensitive at the median lethal concentration (LC_{50}) of chlorantraniliprole.
- The toxicity of indoxacarb (e.g. Steward®) was significantly lower in FAW compared with *H. armigera* and probably represents a naturally higher tolerance to indoxacarb in FAW.
- There was a small but significant reduction in sensitivity to methomyl in FAW larvae compared with *H. armigera.* This is consistent with the detection of genetic markers for carbamate resistance in FAW. However, moths of FAW remain fully susceptible to methomyl.
- FAW is 50-80 times less sensitive to the synthetic pyrethroids (SP) alpha-cypermethrin and gamma-cyhalothrin compared with susceptible *H. armigera*. Therefore, based on our experience with *H. armigera* with similar levels of SP resistance, it is highly unlikely that field rates of these insecticides will control FAW, even under optimal spray conditions.
- There is strong evidence to suggest metabolic (not target site) resistance to SP in FAW. Metabolic resistance is an important mechanism which is also known to confer very high levels of SP resistance in *H. armigera*.

Table 1. Summary of permits for fall armyworm control as of January 2021.

Active Constituent	MOA Group	Permit number
Methomyl	1A	PER89279, PER89293, PER89400, PER89330
Alpha-cypermethrin	3A	PER89279, PER85447, PER89425, PER89330
Gamma-cyhalothrin	3A	PER89358
Spinetoram	5	PER89241, PER89331, PER89327, PER89284, PER89390
Spinosad	5	PER89870
Emamectin benzoate	6	PER89285, PER89263, PER89300, PER89344, PER89371, PER89330
Indoxacarb	22A	PER89306, PER89279, PER89278, PER89311, PER89530, PER89286, PER90374, PER89330
Chlorantraniliprole	28	PER89290, PER89366, PER89281, PER89353, PER89384, PER89259, PER89354, PER89457, PER89330

Implications for management

High levels of metabolic resistance to SP and presence of genetic markers for resistance to carbamates and organophosphates indicate that broad-spectrum insecticides are unlikely to provide effective control of FAW. Given the levels of resistance to broad-spectrums, growers are strongly advised to avoid using these chemical groups and instead consider adopting IPM strategies which help optimise the cost of controlling FAW by taking advantage of natural enemies present in crops.

High levels of susceptibility to selective insecticides such as emamectin benzoate and spinetoram indicate these insecticides will be effective options for management. The Group 28 insecticide chlorantraniliprole is also likely to provide effective control. However, control may be marginal at rates below the full field rate of this insecticide.

A natural tolerance to indoxacarb in FAW suggests this insecticide may not provide effective control in crops with high insect pressure. However, indoxacarb may be useful for achieving population suppression in low pressure situations and for providing an additional rotation option for resistance management.

As with any insect pest, there is considerable potential for further selection of resistance in FAW to selective insecticides if usage increases. Overuse of selective insecticides could also threaten Helicoverpa resistance management if there is an increase in the frequency of sprays in crops where the two species occur together.

As a first step toward pre-emptive management of resistance to pivotal selective insecticides used in the management of FAW, NSW DPI has developed resistance screening procedures to increase capacity for detecting field resistance in FAW. Diagnostic concentrations of indoxacarb, chlorantraniliprole, emamectin benzoate and spinetoram have now been established for detecting future changes in resistance to these insecticides which will be a critical component of future resistance management strategies that are developed for FAW.

Although there is currently no resistance management strategy (RMS) for FAW, the key principles of resistance management and IPM should be applied when making spray decisions. This includes regular monitoring to identify early outbreaks, timely applications of selective insecticides on above threshold populations, and rotating selective insecticides with different modes of action.

Following these guidelines will help to optimise the cost of applications and control of FAW while making the most of natural enemy populations present in the crop, the benefits of which will be destroyed by broad-spectrum insecticides that are unlikely to provide effective control.

Detecting FAW in sorghum and corn

FAW are active across the northern grains region, but inland central and southern Qld have not experienced continuous population build up in crops since the first immigration of moths in September – October. Infestations are typically patchy across fields with 'hotspots' of heavily impacted plants. Evidence of FAW infestation appear at any stage of crop growth, but no reports of significant crop loss have been recorded to date.

One of the major contributors to this continuing low pressure and minor crop damage is probably the very high natural enemy (beneficial) impact on FAW. There are a number of common parasitoids and predators we are familiar with as natural enemies of helicoverpa, armyworms and loopers that are being observed attacking FAW. These include the larval parasitoid wasp *Cotesia* sp. (possibly *Cotesia ruficrus*) and the spined predatory bug (*Oechalia schellenbergii*). One of the other common, but more difficult to see, natural enemies is *Trichogramma* (egg parasitoid). In north Queensland, *Trichogramma* egg parasitism has been very common in our trial plots. When *Trichogramma* find a FAW egg mass, all eggs are parasitised, or at least the vast majority.

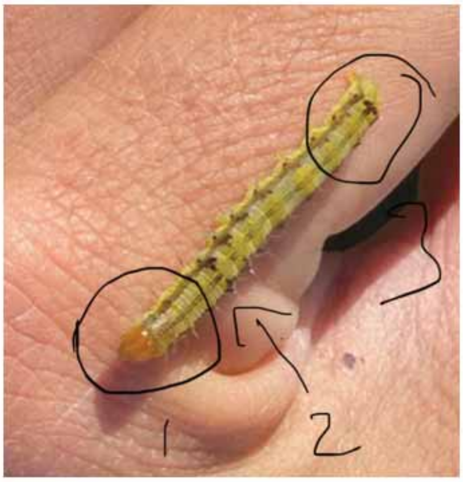
Figure 1. The most commonly observed natural enemies attacking FAW to date. (Left to right). Group of white, fluffy pupal cocoons of the larval parasitoid, *Cotesia* sp. The spined predatory shield bug making a meal of a FAW larva. A FAW egg mass that has been parasitised by *Trichogramma*. The small exit holes in the eggs are visible where the wasps have emerged.

Larval identification basics

It is very likely that both helicoverpa and FAW will be present in most maize and sorghum crops, as well as native armyworm (common, northern) in some crops. There are identification resources on the Beatsheet.

FAW and helicoverpa are very similar. Once you have had a bit of experience with FAW, the differences will be obvious, but until then it is a struggle to work out which is which, and why. These images below have been sent in for checking – to see if they are FAW or helicoverpa. If in doubt, please to send photos to Melina Miles (Melina.Miles@daf.qld.gov.au) or Hugh Brier (hugh.brier@daf.qld.gov.au).

The annotation in Figure 2 (excuse the amateur attempt) identifies the features that determine whether they are helicoverpa or FAW. As with all insects and characters, there is variation between individuals. Darker FAW larvae can look to have darker spots etc. In the case of Figure 2 below, this is a *Helicoverpa armigera*.



1. The collar (the narrow shield behind the head) is a different colour to the head = helicoverpa. In FAW, the head and the collar are generally the same colour.
2. The 'saddle' is visible on this specimen = Helicoverpa armigera
3. The spots at the rear end, although in a square (4 dots in a square) are the same colour and 'intensity' as the spots along the rest of the body. In a FAW larva this size, the spots on the rear are much more prominent than other spots on the body.

Figure 2. Identification points differentiating *Helicoverpa armigera* and fall armyworm (picture is *Helicoverpa armigera*)

Figure 3. Helicoverpa and FAW larvae (3^{rd}-4^{th} instar) are very difficult to tell apart, until you have had some experience with them. These images illustrate how similar the two are at this stage. Helicoverpa (left image), FAW (right).

Figure 4. Large FAW larvae, like this one, start to look greasy and less hairy than younger FAW or helicoverpa. In this image you can see how much darker and more raised the square of spots is on the rear end. This is very characteristic of late instar FAW.

Recognising FAW damage

What will be most obvious, when FAW are present, is the amount of damage that they cause to the leaves. We are used to seeing some windowing from helicoverpa, but largely they do very little damage to the developing whorl. The worst of it is usually 'shot holes' in the expanded leaves (see Figure 5).

Figure 5. (Left) Helicoverpa damage to sorghum tends to be minor windowing evident in expanded leaves. The whorl is largely unaffected. Photos: Rory Kerlin. (Right) Shotgun holes in leaves, typical of helicoverpa damage.

In contrast, FAW larvae feeding in the whorl cause significant damage to the whorl. This will be evident, not only by examining the whorl, but from the large holes in the expanding leaves as a result of the amount of damage caused in the whorl (Figure 6)

Figure 6. FAW damage to sorghum, showing the extensive damage to the whorl, and the large holes evident as leaves expand. The last image shows a sorghum plant with very characteristic damage to the whorl where the terminal end of the whorl has been chewed off. As a result, the whorl and emerging leaves are cut (or break) off flat.

FAW larvae causing extensive damage in the whorl are generally medium and large larvae. Smaller larvae can be present in the whorl, and unrolling the whorl may expose multiple larvae that have segregated themselves from each other in the different layers.

Feeding by very small and small FAW larvae that have recently emerged from eggs (windowing) is also an indication of FAW activity in the crop. Keep in mind that windowing will persist on leaves long after the larvae have moved to the whorl or died. With experience, new and old feeding damage can be distinguished (Figure 7).

Figure 7. (Left) Feeding damage caused by newly hatched FAW larvae. The remains of the egg mass (moth scales) is just visible on the RHS of the feeding damage. (Right) Windowing caused by small FAW not yet established in the whorl.

The other thing you will notice with FAW, that you don't see so much with helicoverpa, is the amount of poo/frass they produce. Huge amounts of it that fills the whorl. It is wet and sloppy, not dry pellets like helicoverpa and native armyworm species tend to produce (Figure 8).

Figure 8. Comparison of frass (poo) of helicoverpa and FAW. Helicoverpa poo (left) is dry. FAW poo is sloppy and messy, and there is a lot of it! (right).

FAW moths are often sighted resting in crops, in similar locations to Helicoverpa moths (Figure 9). Both male and female moths have been observed resting in whorls particularly.

Figure 9. Female FAW moth.

Scouting for eggs can feel like looking for a needle in a haystack. There is so much leaf area in a crop of sorghum or maize. Our experience in north Qld so far, has been that eggs are often laid in about the same place on plants. On small plants, they are often on the undersides of the leaves, towards the base (Figure 7). Eggs are not always covered in fluffy scales from the moth as in Figure 10a and 10b. Some are covered in scales like the egg mass in Figure 10c. We have found it a bit easier to find eggs on the underside of leaves if you are checking in the early morning or late afternoon. At these times of the day the eggs on the underside of the leaf can be silhouetted (Figure 10d), meaning that you can walk along rows looking for the shadows, rather than turning leaves over constantly.

Figure 10. Location of FAW eggs are laid on the underside of leaves in young crops.

Damage to establishing crops can include plant death

In establishing crops, FAW will not feed only on leaves. They can also burrow into the base of young plants, resulting in the death of the plant. Where we have seen this happen it has been where there are large larvae which have consumed most of the plant and then taken shelter during the day in the soil. These large larvae will sit under the soil surface feeding on the base of the seedling plants (Figure 11). For this reason, it is important that emerging crops are checked at least weekly. If a high percentage of plants have FAW larvae, and the larvae are medium or larger, there is a risk that they could start to do this kind of damage. Once plants have 6-8 leaves, there is more leaf and less likelihood of this type of damage occurring.

Figure 11. Large FAW larvae sheltering under the soil surface and feeding on the base of seedlings. Damage caused by this feeding. Larvae can burrow up inside the stem too.

If the damage observed in a young crop seems disproportionately high for the number, or size, of larvae being observed on plants, check for large larvae around the base of plants. It is possible that large, very damaging larvae, are moving up onto plants at night to feed, but are not there to be sampled during the day.

References

The Beatsheet (www.thebeatsheet.com.au) has resources on FAW and other caterpillar identification, pheromone trap catch data for Qld and NSW, webinars, access to a development model for insect pest species, how-to videos and regular updates on regional pest issues.

APVMA PUBCRIS database (https://portal.apvma.gov.au/permits) (search for fall armyworm) to find current permits for control options.

Acknowledgements

CRDC, NSW DPI and the Queensland Government have provided financial support for the R, D and E presented in this paper. We also thank agronomists and growers who have provided FAW collections for resistance testing and/or provided access to crops on their farms, and to the QDAF farm staff at the Ayr Research Station.

Unless photographer is specifically identified, all photographs by Melina Miles.

Contact details

Dr Melina Miles
Department of Agriculture and Fisheries, Toowoomba
Ph: 07 45294169 / 0407113306
Email: melina.miles@daf.qld.gov.au

Dr Lisa Bird
NSW Department of Primary Industries, Tamworth
Ph: 02 67631128 / 0438623906
Email: lisa.bird@dpi.nsw.gov.au

® Registered trademark

Herbicide resistance survey results summer annual weed species: fleabane, feathertop Rhodes grass, barnyard grass, sowthistle, windmill grass

Michael Widderick[1], Michael Walsh[2], Adam Jalaludin[1] and John Broster[3]

[1] Queensland Department of Agriculture and Fisheries, Toowoomba, Qld
[2] University of Sydney, Camden, NSW
[3] Charles Sturt University, Wagga Wagga, NSW

Key words

herbicide resistance, glyphosate resistance, summer weeds, northern grains cropping region

GRDC codes

UCS00024, US00084

Take home messages

- Glyphosate resistance is common in the northern grain cropping region: flaxleaf fleabane (100% of populations), feathertop Rhodes grass (68%), windmill grass (58%), awnless barnyard grass (36%), common sowthistle (14%)
- Haloxyfop (Group A) resistance was confirmed in one population of feathertop Rhodes grass and this resistance is likely to become more common under current Group A herbicide use patterns for fallow grass weed control
- 2,4-D amine controlled all fleabane and sowthistle populations, including those that survived glyphosate
- Group A herbicides haloxyfop, propaquizafop and clethodim gave good control of the remaining summer grass weeds and all feathertop Rhodes grass populations were susceptible to paraquat
- Multiple chemical and non-chemical weed control options need to be used to delay herbicide resistance and retain the usefulness of herbicides for weed control.

Introduction

To better understand the extent of herbicide resistance in the dominant summer annual weed species of the northern grain cropping region, a weed seed collection survey was conducted during the 2016-17 and 2017-18 summer grain cropping seasons in summer cropping regions (Qld and northern NSW).

This paper outlines the survey and provides results on which weeds were most common and the herbicide resistance status of key summer weeds including common sowthistle (*Sonchus oleraceus*), flaxleaf fleabane (*Conyza bonariensis*), awnless barnyard grass (*Echinochloa colona*), feathertop Rhodes grass (*Chloris virgata*) and windmill grass (*Chloris truncata*).

Material and methods

Random field surveys were conducted across the northern grain cropping region by Queensland Department of Agriculture and Fisheries in conjunction with Charles Sturt University. Field surveys were conducted in February – March 2017 and February – April 2018. A total of 237 paddocks were surveyed in fields with summer grain crops or in fallow.

Surveying involved collecting seed heads from weeds that were present at sampling, which was timed to coincide with crop maturity. At each collection site, GPS-coordinates were recorded along with crop/fallow situation, presence of target weed species and approximate density. Other weed species were also noted to identify weeds for inclusion in future surveys.

Target weeds collected included common sowthistle, flaxleaf fleabane, awnless barnyard grass, feathertop Rhodes grass and windmill grass. These weeds were screened for resistance to the herbicides commonly used for their control (Table 1).

A winter crop survey was also conducted at crop maturity in 2017 and 2018 and the presence and frequency of target summer weed species that were found in winter crops at harvest are presented (Table 2).

Seed germination and herbicide resistance screening

Weed seeds were grown in the growing seasons following their collection. Sowthistle were grown from late March to late November while fleabane and the summer grasses were grown from late November to mid-May.

Plants were grown until they reached three to five leaf stage. They were then treated at the recommended label rate for each herbicide (Table 1) with the appropriate adjuvant (if required). Herbicides were applied using an enclosed cabinet sprayer. For weeds that did not have registration for a particular herbicide, recommended rate for the closest relative weed was used. Assessment for survivors was carried out at 21 days after treatment (DAT). Plants were considered surviving if there were actively growing tillers (grass weeds) or regrowth from the growing point (broadleaf weeds). A population was classed as resistant if at least 20% of plants survived treatment.

Table 1. Herbicides and rates used for screening various weed species for resistance.

Weed species	Herbicide (note rates are of active ingredient)
Common sowthistle	Glyphosate (729 g ae/ha) 2,4-D amine (1050 g ai/ha) Velocity® (bromoxynil 210 g ai/ha + pyrasulfotole 37.5 g ai/ha) Chlorsulfuron (15 g ai/ha) †
Flaxleaf fleabane	Glyphosate (729 g ae/ha) † 2,4-D amine (1050 g ai/ha)
Feathertop Rhodes grass	Glyphosate (729 g ae/ha) † Haloxyfop (78 g ai/ha) Clethodim (90 g ai/ha) Paraquat (400 g ai/ha)
Awnless barnyard grass	Glyphosate (729 g ae/ha) Propaquizafop (60 g ai/ha) Clethodim (90 g ai/ha) Imazapic (96 g ai/ha) ** (post-emergence application)
Windmill grass	Glyphosate (729 g ae/ha) Clethodim (90 g ai/ha) ***

†Not registered for control of this weed species
** Used alone as post-emergence. Note that imazapic is only registered for stand-alone use against these weeds as a pre-emergent application. ***As per APVMA permit PER89322 (but without the double knock)

Results and discussion

Weed occurrence

In the northern region, common sowthistle was the most commonly occurring weed, present in both summer and winter crops at harvest time (Table 2). The summer grasses feathertop Rhodes grass and awnless barnyard grass were present in 20% of Queensland fields surveyed but only 3 and 5% respectively of fields surveyed in NSW. Flaxleaf fleabane was less common in Queensland, being present in 11% of summer and winter fields surveyed, compared to 15% of surveyed paddocks in NSW. Sweet summer grass (*Brachiaria eruciformis*), which was not a target weed, was present in both summer and winter surveys occurring in 9% of fields. These fields were all in Central Queensland.

Table 2. Incidence and frequency of occurrence of major summer weed species in paddocks surveyed during the weed seed collection survey of summer and winter cropping paddocks at crop maturity in 2017 and 2018. Expressed as both number and percent of fields surveyed in the northern grain cropping region.

Weed species	Queensland		New South Wales	
	No. of fields	% of fields	No. of fields	% of fields
Common sowthistle	97 (W/S)	26	104 (W)	33
Feathertop Rhodes grass	75 (W/S)	20	12 (S)	3
Awnless barnyard grass	34 (S)	20	17 (W/S)	5
Flaxleaf fleabane	42 (W/S)	11	47 (W/S)	15
Sweet summer grass	34 (W/S)	9	0	0
Liverseed grass	8 (S)	5	5 (S)	2
Windmill grass	2 (S)	1	15 (S)	6

W = winter crop survey, S = summer crop survey.

Common sowthistle resistance

Screening of 197 viable sowthistle populations with glyphosate and 2,4-D amine detected 14% were resistant to glyphosate while 100% were susceptible to 2,4-D amine (Table 3). The glyphosate resistant populations were distributed throughout the northern region (Figure 1). Screening of 136 populations with Velocity® and chlorsulfuron showed that Velocity® was able to control all populations while 95% of populations were resistant to chlorsulfuron (Table 3).

Table 3. Number of collected populations of common sowthistle and flaxleaf fleabane screened for resistance and the frequency of these populations that were resistant to glyphosate, 2,4-D amine, chlorsulfuron and Velocity®.

Weed species	Number of populations tested	Glyphosate (%)	2,4-D amine (%)	Chlorsulfuron (%)	Velocity® (%)
Common sowthistle	197	14	0		
	136			95	0
Flaxleaf fleabane	61	100	0	n/a	n/a

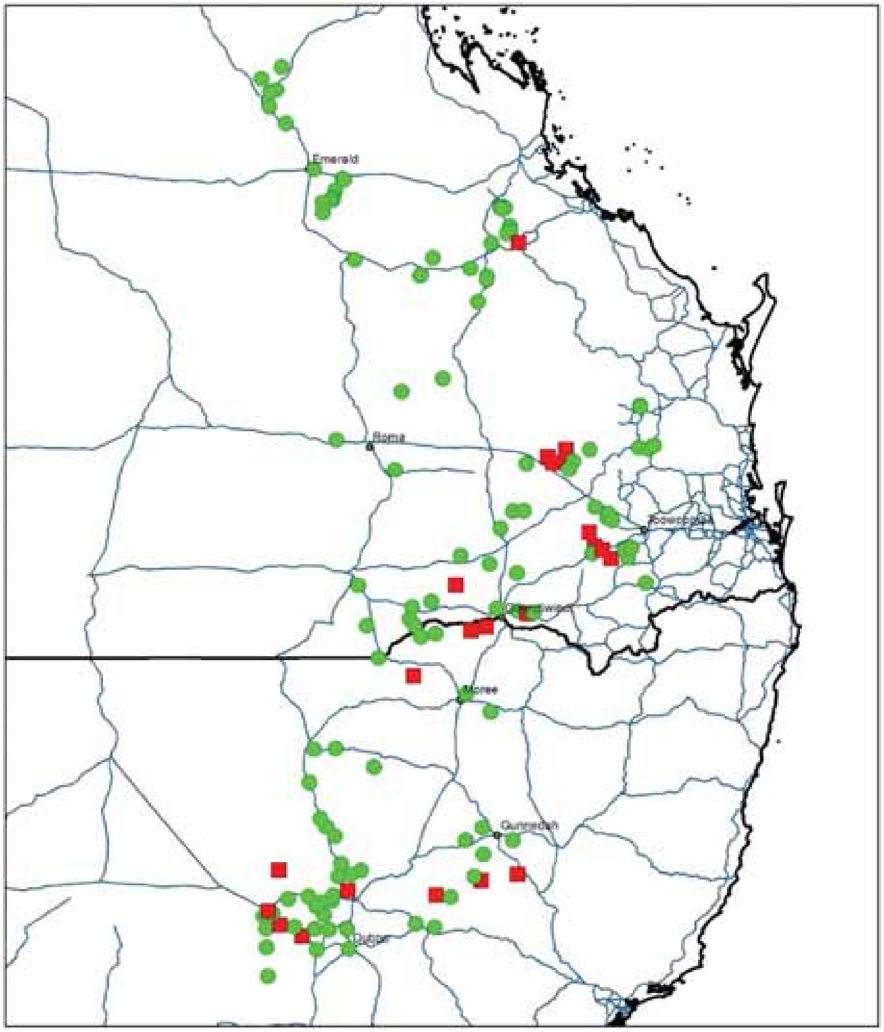


Figure 1. Distribution of glyphosate resistant and susceptible common sowthistle populations across the northern grain cropping region. Red squares represent resistant populations while green circles represent susceptible populations.

Flaxleaf fleabane resistance

Of the 89 fleabane populations collected across the northern region, only 61 were viable (Figure 2). Glyphosate alone is not registered for fleabane control and none of the populations screened were controlled by this herbicide (Figure 2). None of the populations survived treatment with 2, 4-D amine (Table 2).

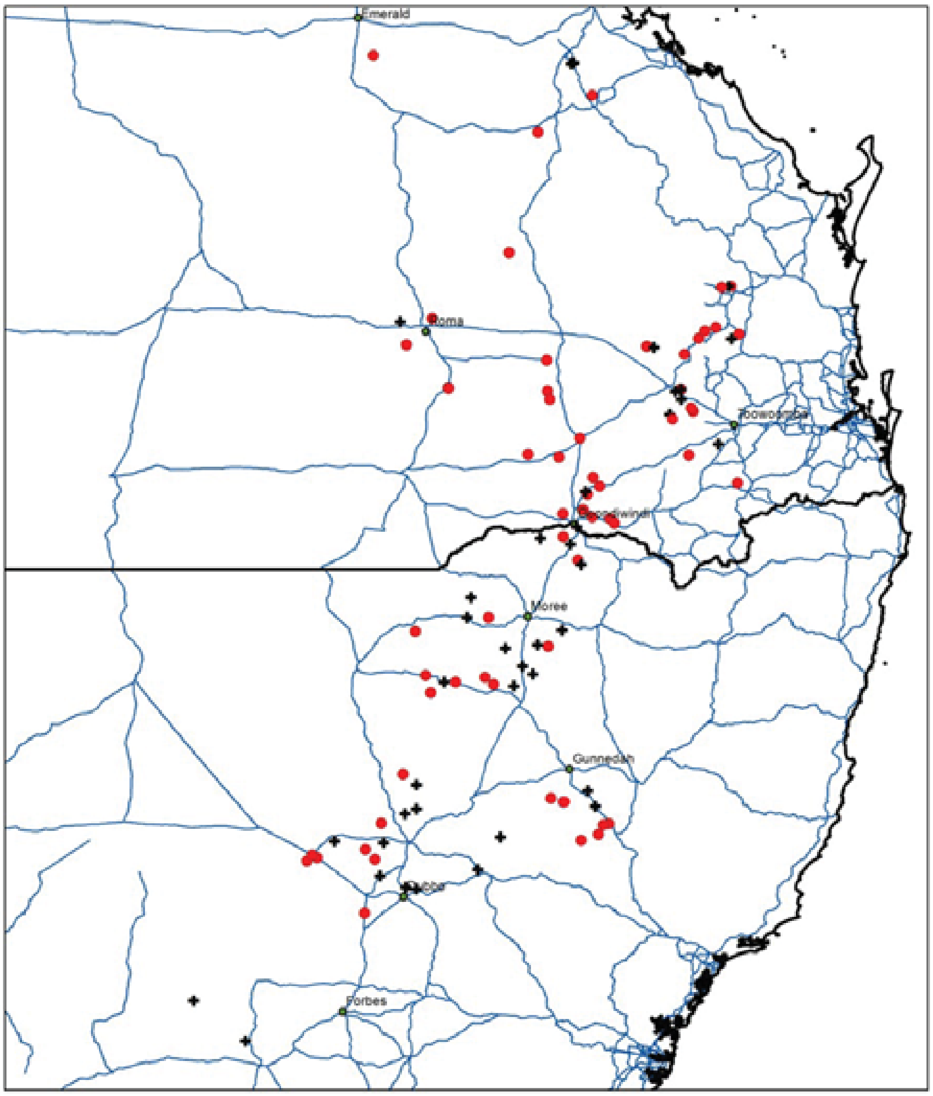


Figure 2. Distribution of glyphosate resistant flaxleaf fleabane populations across the northern grain cropping region. Red circles are resistant populations, while black crosses represent non-viable populations.

Feathertop Rhodes grass resistance

Screening of 62 viable populations revealed 68% survived the glyphosate target rate (noting that glyphosate is not registered for FTR control (Figure 3, Table 4). One population survived treatment with haloxyfop. This population was from north of Biloela in Central Queensland. All populations were controlled with clethodim and paraquat.

Table 4. Number of populations of feathertop Rhodes grass screened for resistance and the frequency of these populations that were resistant to glyphosate, haloxyfop, clethodim and paraquat.

Weed species	Number of populations tested	Glyphosate (%)	Haloxyfop (%)	Clethodim (%)	Paraquat (%)
Feathertop Rhodes grass	62	68	2	0	0

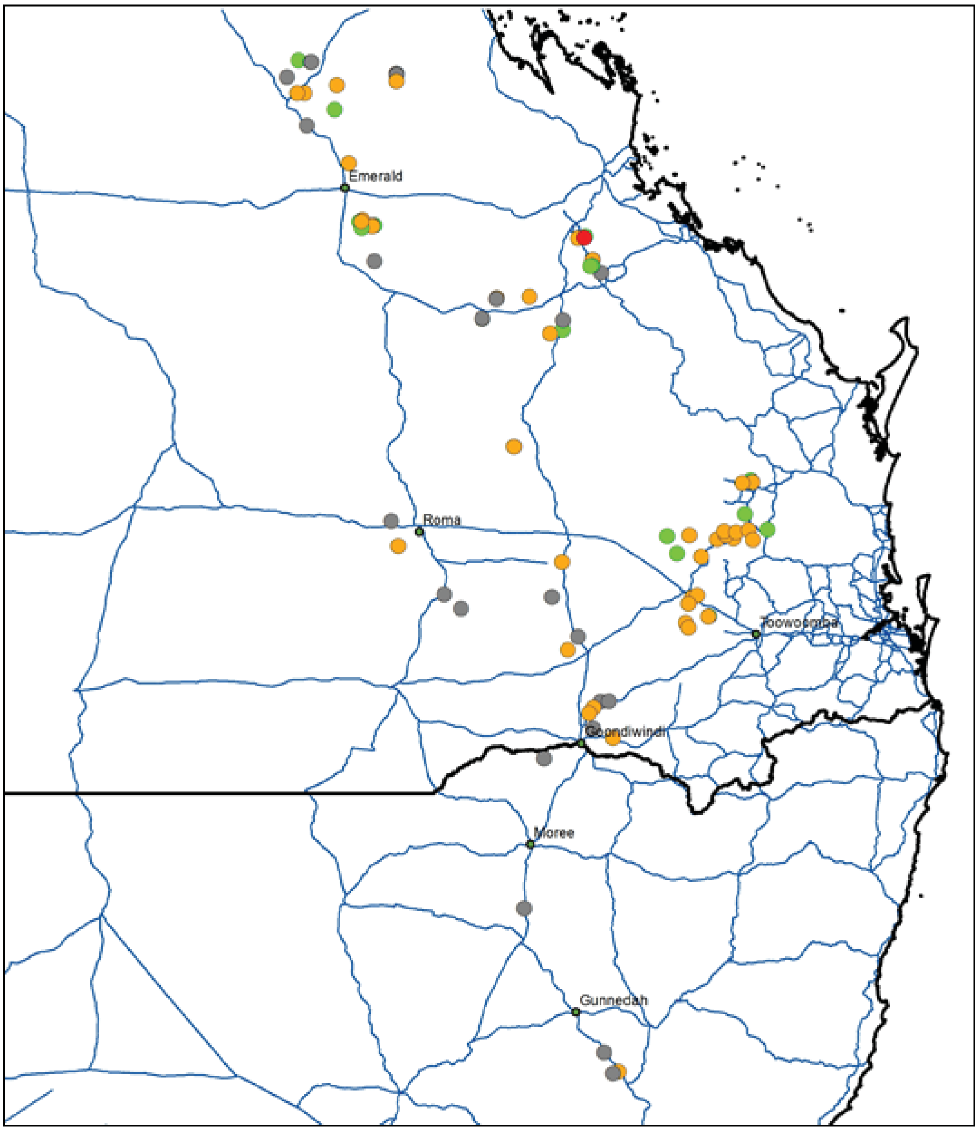


Figure 3. Distribution of feathertop Rhodes grass resistances across the northern grain cropping region. A red circle represents a glyphosate and haloxyfop resistant population, orange circles represent glyphosate resistant populations that are susceptible to haloxyfop, green circles represent populations susceptible to all herbicides tested, and grey circles represent populations that were not viable.

Awnless barnyard grass resistance

Screening of 42 viable populations revealed 36% were resistant to glyphosate (Figure 4, Table 5), while all populations were susceptible to propaquizafop, clethodim and imazapic.

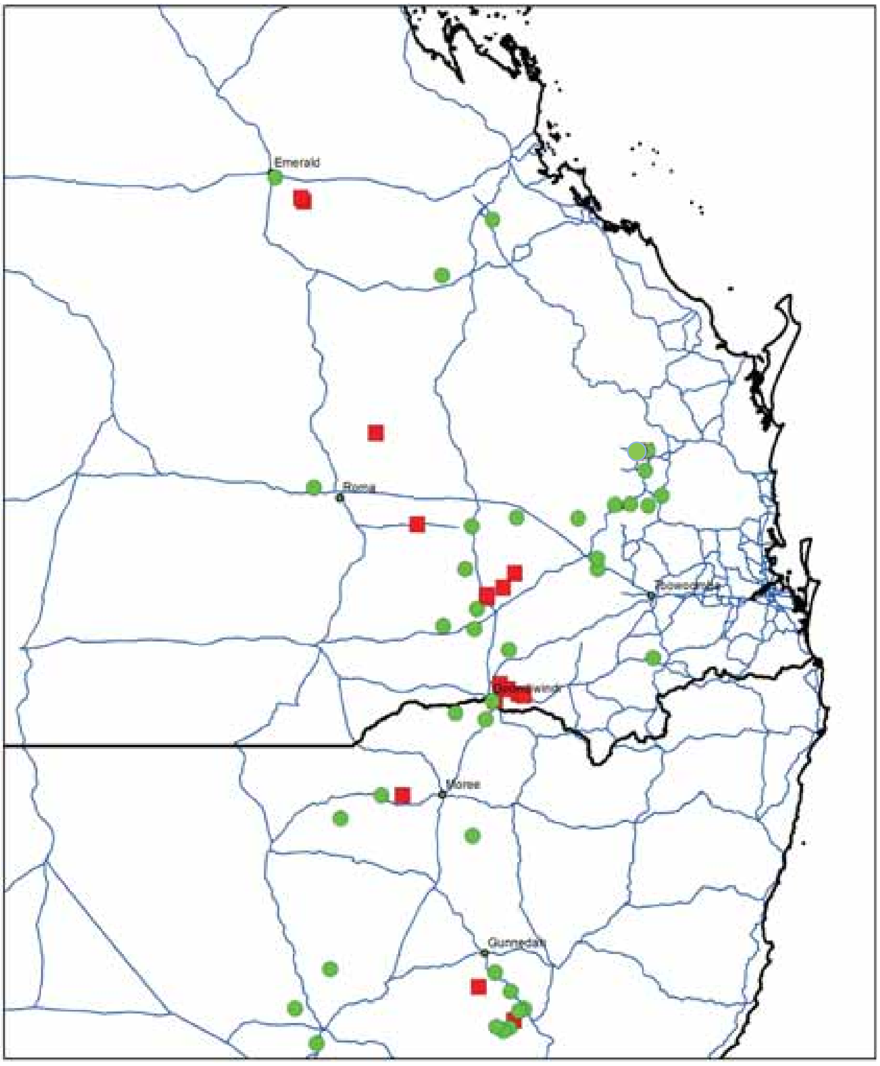


Figure 4. Distribution of glyphosate resistant and susceptible awnless barnyard grass populations across the northern grain cropping region. Red squares represent glyphosate resistant populations while green circles represent susceptible populations.

Table 5. Number of collected populations of awnless barnyard grass and windmill grass screened for resistance and the frequency of these populations that were resistant to glyphosate, propaquizafop, clethodim and imazapic.

Weed species	Number of populations tested	Glyphosate (%)	Propaquizafop (%)	Clethodim (%)	Imazapic (%) *
Awnless barnyard grass	42	36	0	0	0
Windmill grass	12	58	n/a	0	n/a

*Imazapic was applied post-emergence as a foliar application, and not as a pre-emergence soil residual treatment

Windmill grass resistance

Screening of 12 viable populations revealed that more than half (58%) had evolved resistance to glyphosate (Table 5). All populations were controlled by clethodim. As only a small number of windmill grass populations were screened for resistance, results cannot be considered representative of windmill grass throughout the northern grain cropping region.

Discussion

The survey has shown that glyphosate resistance is common in the northern grain cropping region for several key summer weed species including common sowthistle and awnless barnyard grass. Although glyphosate applied alone is not registered for control of feathertop Rhodes grass or flaxleaf fleabane, herbicide screening results showed overwhelmingly that this herbicide is ineffective on these species. Glyphosate is still the main stay of fallow weed control in Qld grain cropping systems and has the benefit of controlling a range of weed species that may be present together during a fallow. However, results of this screening have shown glyphosate efficacy is likely to be less reliable across the region due to resistance. Therefore, other weed control options are required.

The resistance screening identified a range of commonly used herbicides that are still effective on most populations of target summer weed species. 2,4-D amine controlled all fleabane and sowthistle populations tested, including those that survived glyphosate. Group A herbicides haloxyfop, propaquizafop and clethodim gave good control of the summer grass weeds, with only one feathertop Rhodes grass population surviving haloxyfop treatment.

It is important to continue to survey for herbicide resistance as the occurrence and distribution is likely to change over time. The GRDC has recently invested in a new national herbicide resistance survey (2020 – 2023) to provide more up-to-date information on herbicide resistance nationally. In the northern grain cropping region, both winter and summer crop surveys will be undertaken. Target weed species are common sowthistle, awnless barnyard grass, feathertop Rhodes grass, fleabanes, wild radish, sweet summer grass and wild oats. If you would like to participate in this survey by providing permission for researchers to enter your property for weed seed collection, please contact the author of this paper.

Acknowledgements

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.

Contact details

Michael Widderick
Principal Research Scientist
Queensland Department of Agriculture and Fisheries
Leslie Research Facility, Toowoomba
Ph: 07 4529 1325
Email: Michael.widderick@daf.qld.gov.au

Herbicide resistance and optimising glyphosate- Dalby

Peter Boutsalis[1,2] & Christopher Preston[1]

[1] School of Agriculture, Food & Wine, University of Adelaide
[2] Plant Science Consulting P/L

Keywords

optimising glyphosate, barnyard grass, wild oat, resistance, environmental stress, herbicide application

Take home messages

- Glyphosate performance can be optimised through attention to application variables such as temperature, plant stress and formulation.
- Wild oat resistance levels show effective herbicide options are available.
- Resistance testing is important to identify effective herbicides.

Evolution of glyphosate resistance

Glyphosate was first registered in the 1970's and rapidly became the benchmark herbicide for non-selective weed control. The first weed species to develop resistance to glyphosate was annual ryegrass that occurred in an orchard in southern NSW (Powles *et. al.* 1998). Only a few cases of resistance were detected in the following decade . The fact that it required decades of repeated use before resistance to other species was confirmed indicated that the natural frequency of glyphosate resistance was initially very low. At the current time there are over a dozen species that have developed resistance to glyphosate in Australia (https://www.croplife.org.au/resources/programs/resistance-management/herbicide-resistant-weeds-list-draft-3/). The most important species are ryegrass, awnless barnyard grass, feathertop Rhodes grass, liverseed grass, flaxleaf fleabane and sowthistle.

Resistance is generally not 'black and white' with resistance levels differing between populations of resistant weeds and even between individuals of the same population. Differences in the level of resistance have been detected in barnyard grass (Figure 1).

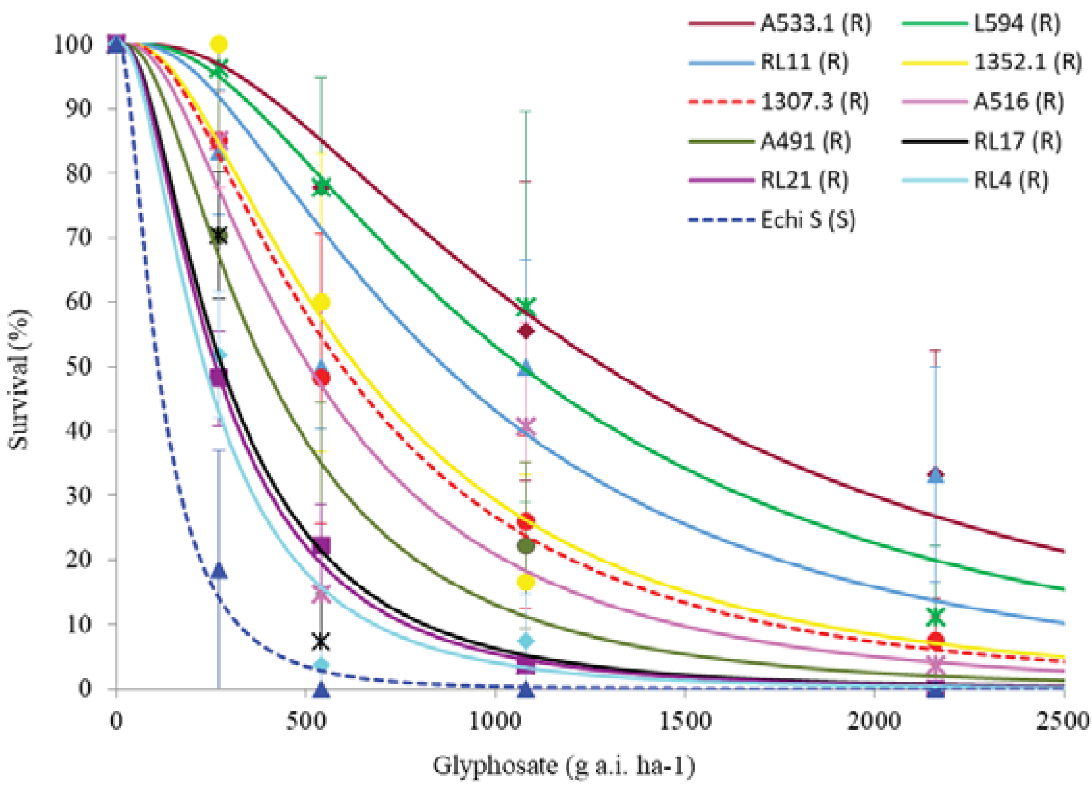


Figure 1. Response of 10 resistant awnless barnyard grass biotypes to increasing rates of glyphosate (The University of Adelaide). 'Echi S' is the susceptible control population.

Increasing glyphosate resistance

There are several contributing factors for the increasing incidence in glyphosate resistance, with generally more than one factor responsible. These include:

1. Reducing rates or using poor quality formulations, resulting in sub-lethal control (Figure 2)
2. Mixing glyphosate with too many other active ingredients resulting in antagonism, particularly in low water volumes
3. Using low quality water, particularly hard water. Glyphosate is a weak acid and binds to positive cations (i.e. magnesium, calcium and bicarbonate) that are in high concentration in hard water (i.e. >200 ppm)
4. Applying glyphosate during periods of high temperature and low humidity, resulting in the rapid loss of glyphosate in solution from leaf surfaces thereby reducing absorption
5. Translocation of glyphosate in stressed plants can be reduced (Figure 3). Maximum glyphosate efficacy relies on translocation to the root and shoot tips. While this occurs readily in small seedlings, in larger plants, glyphosate is required to translocate further to the root and shoot tips to provide high levels of control
6. Shading effects reducing leaf coverage resulting in sub-lethal effects
7. As glyphosate strongly binds to soil particles. Application onto dust covered leaves can reduce efficacy
8. Application factors such as speed and nozzle selection, boom height can reduce the amount of glyphosate coverage
9. A combination of the above factors can reduce control thereby increasing the selection for resistance.

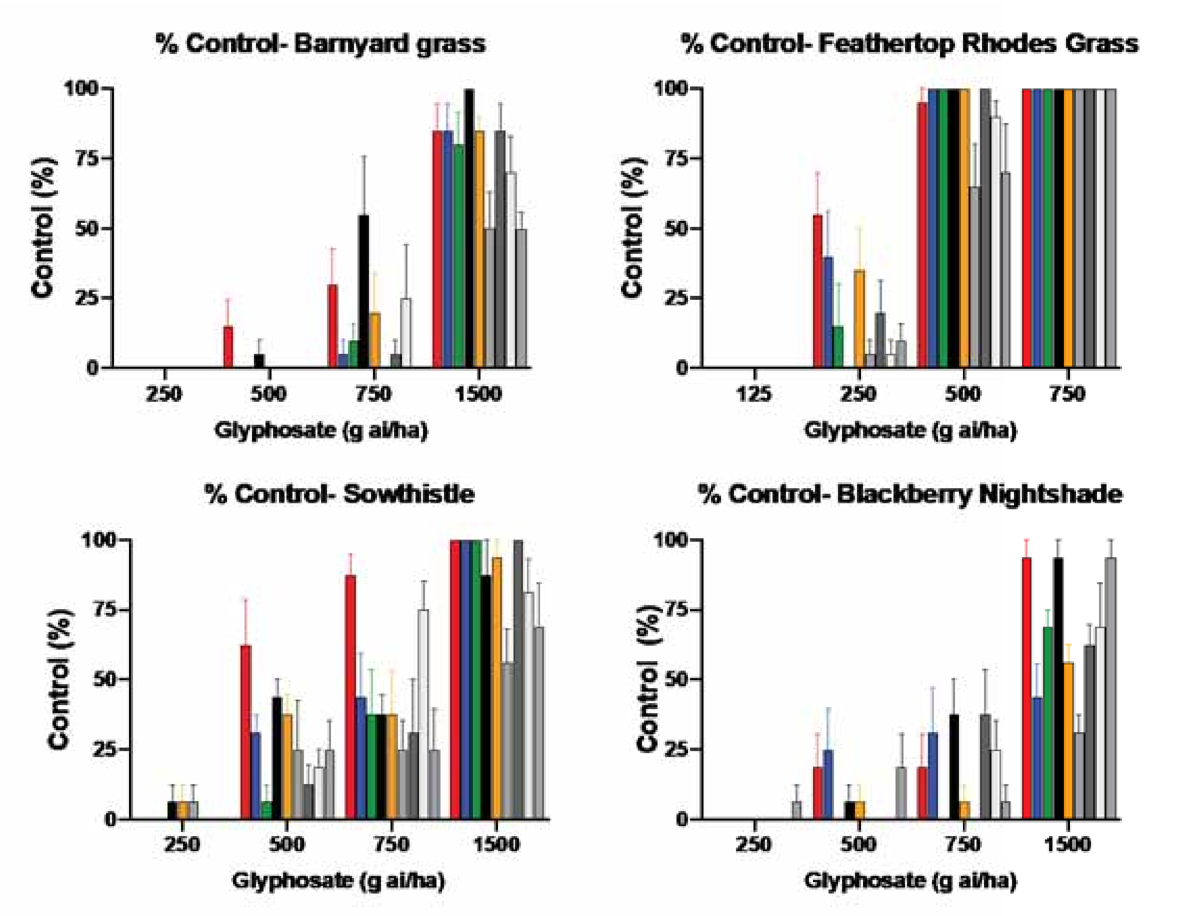


Figure 2. Control of 4 summer weed species with 9 different glyphosate formulations. (Plant Science Consulting).

Optimising glyphosate performance

The selection of glyphosate resistance can be minimised by considering the points above. A number of important pathways to improve glyphosate performance include:

Avoid applying glyphosate under hot dry conditions

Reduced control can occur if plants are water stressed. In an outdoor summer pot trial, the effect of water stress on glyphosate activity on barnyard grass was investigated. A sub-set of plants were water stressed 2 days prior to application with glyphosate. Plants at three growth stages were included. Control of stressed plants (drought treatment) was significantly less than of non-stressed plants at all growth stages (Figure 3).

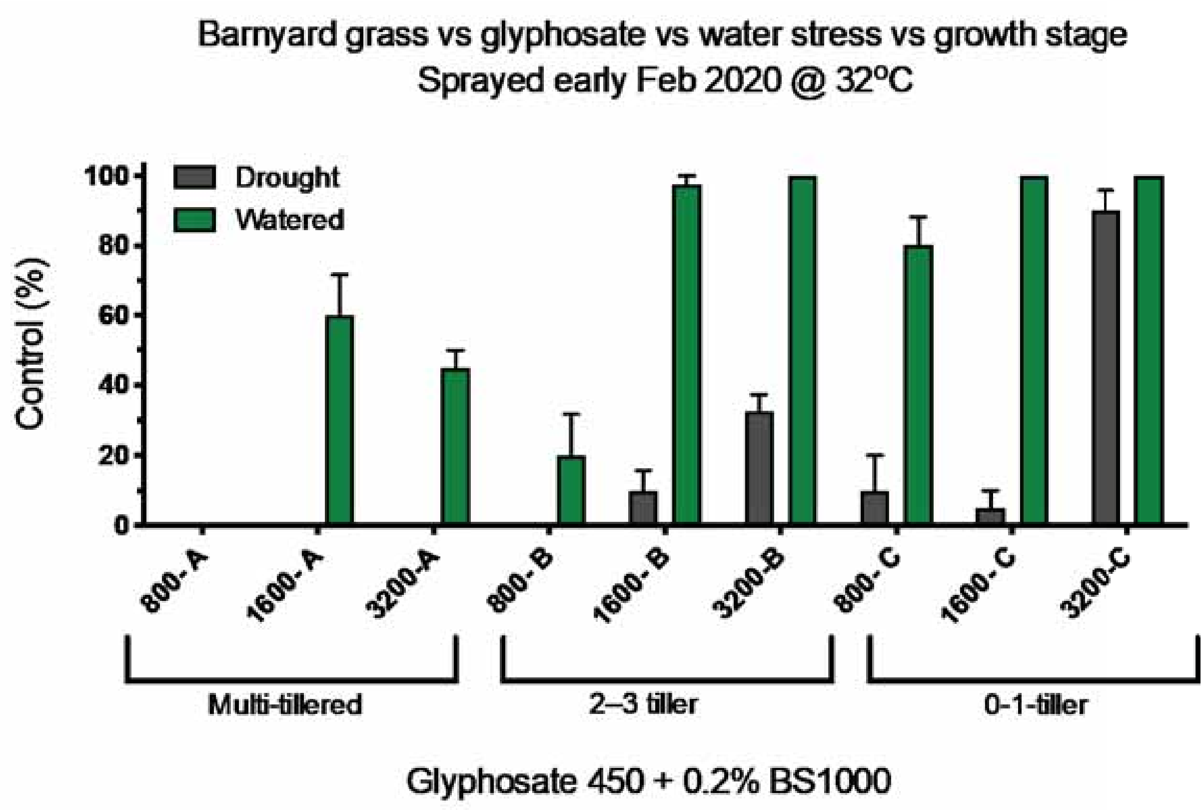


Figure 3. The effect of three rates of glyphosate on three growth stages of awnless barnyard grass, half not water-stressed and the other half water-stressed (Plant Science Consulting).

The effect of temperature on control of glyphosate resistant sowthistle from NSW was recently investigated. Initial trials have confirmed greater control with glyphosate at lower temperatures, particularly of resistant biotypes (Table 1). These findings suggest that applying glyphosate at lower temperatures can improve control of glyphosate resistant sowthistle. At lower temperatures glyphosate remains in liquid form on plant surfaces longer leading to greater uptake, particularly under higher humidity. This allows more hours on the leaf for glyphosate to penetrate. Maximising glyphosate uptake is therefore likely to improve weed control and factors such as lower temperature and higher humidity influence uptake. A pot trial investigating the effect of 20°C and 30°C on glyphosate efficacy identified a higher LD_{50} required at 30°C. This indicates that approximately 2.5x more glyphosate was required to control the resistant populations at 30°C, as opposed to the same populations grown at 20°C

Table 1. Effect of temperature in control of four biotypes of sowthistle from NSW with Glyphosate 540g/L. Data is LD_{50}= dose required to kill 50% of the population.

Biotypes	Resistance level	LD_{50} (g a.i/ha)	
		20°C	30°C
Yellow	strong	439	962
Crocket	strong	389	919
White	weak	132	389
GI	susceptible	135	152

Improving water quality and glyphosate activity by using ammonium sulfate (AMS)

The addition of AMS has several functions. One is to soften water by combining to positively charged ions such as magnesium and calcium common in hard water. The negative charged sulfate ions combine with the positive cations preventing them from interacting with glyphosate and reducing glyphosate solubility and leaf penetration. Additionally, AMS has been shown to independently improve glyphosate performance, as the ammonium ions can work with glyphosate to assist cell entry, increasing uptake and activity. In a pot trial conducted with soft water, ammonium sulfate was shown to significantly improve control of ryegrass with 222 mL/ha (100 g ai/ha) of glyphosate 450 (Figure 4). As a general rule, growers using rainwater (soft) should consider 1% AMS, if using hardwater (i.e. bore, dam) 2% AMS is recommended. The addition of a wetter resulted in a further improvement of herbicide efficacy.

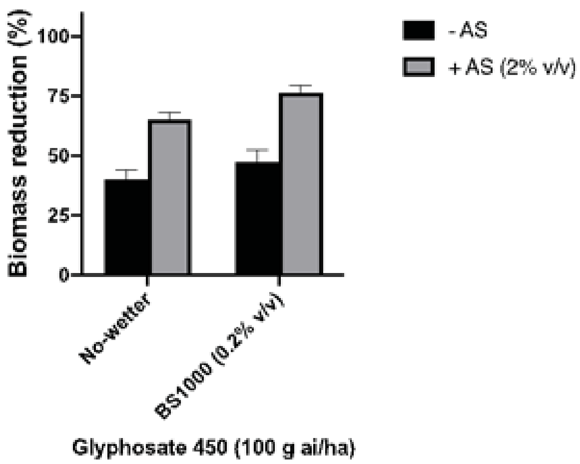


Figure 4. Effect of liquid ammonium sulfate and wetter on glyphosate for ryegrass control. (A sub lethal rate was used to differentiate between treatment differences. Plants were grown and sprayed under optimum conditions).

Wild oat resistance

Over 120 samples of wild oats were received by Plant Science Consulting in 2020 from NSW and Qld, some as plant samples surviving a herbicide application (20 samples) during the cropping season and 100 seed samples currently being resistance tested. The most common mode of action where resistance has been identified to is Group A herbicides such as FOPs, tralkoxydim (eg. Achieve®) and pinoxaden (eg. Axial®). Increasing the rate of these herbicides to the top label rate seldomly improves control. From the plant samples tested over the last winter and spring, 65% were confirmed resistant to Group A herbicides and 35% not resistant, even though many survived a herbicide application in the field. A finding after testing hundreds of samples over the past decade is that clethodim controls wild oats resistant to most other Group A herbicides when used at the top label rate on young (3-leaf to early tillering) and non-stressed plants.

Random weed survey testing has been recently conducted in Qld (Table 2). In 34% of paddocks wild oats was confirmed resistant to clodinafop, with 3% of samples also resistant to Atlantis. A similar result was identified in NSW. This indicates that in a significant number of cases, poor wild oat control with FOP herbicides is due to herbicide resistance. A herbicide resistance test can determine if a poor paddock result is due to herbicide resistance. For more information see the website at the end of this document.

Table 2. Incidence of paddocks containing resistant wild oats as determined by GRDC funded random weed surveying and pot testing by Charles Sturt University, Waggga

Herbicide	QLD	NSW
Clodinafop (eg. Topik)	34	29
Clethodim (eg. Select)	0	1
Mesosulfuron (Atlantis)	3	4
Triallate (eg. Avadex Xtra_	0	0
Glyphosate	0	0
Nr of samples	64	511

Wild oat resistance to Group B herbicides is seldomly detected in resistance tests from Qld and in the case of resistant populations, the level of resistance is usually low. No resistance to triallate has been confirmed. Flamprop-methyl, previously registered as Mataven® and now available as several generic brands, is being included by some growers in their resistant test options.

Discrepancy between resistance testing and paddock failures

In some cases, plants that survived herbicides in the paddock are not resistant. Reasons for the discrepancy between the paddock and a resistance test can include poor application, antagonistic tank mixes, inferior herbicide quality, poor water quality, incorrect adjuvants, or a combination of the above. In addition, as wild oats are particularly prone to stressing by environmental extremes such as frost, heat and drought, herbicide failures can be due to these factors and not herbicide resistance. For this reason, it is of particular importance to have survivors tested for herbicide resistance, either by testing the seed or plants. For more information visit www.plantscienceconsulting.com.au.

Summary

Since the first detection of glyphosate resistance in 1998 the number of resistant cases in several species has rapidly increased. Decades of strong selection pressure resulting from repeated use of glyphosate, coupled with application under suboptimum conditions and poor application has played a major role. Understanding the variables that affect glyphosate is important to optimise control. Resistance testing verifies that poor wild oat control in the field is not always due to herbicide resistance.

Acknowledgements

The information from Figure 1 is from research conducted at the University of Adelaide.

References

Powles SB, Lorraine-Colwill DF, Dellow JJ, Preston C. 1998. Evolved resistance to glyphosate in rigid ryegrass (*Lolium rigidum*) in Australia. *Weed Science.* 46:604–607

Contact details

Peter Boutsalis
Plant Science Consulting P/L
University of Adelaide, Waite Campus, Glen Osmond SA 5064
Herbicide resistance testing website: www.plantscienceconsulting.com.au
@PBoutsalis
Email: peter.boutsalis@adelaide.edu.au
Email: info@plantscienceconsulting.com.au

® Registered trademark

Persistence and retention of weed seeds in the northern grain region of Australia: implications for management

Bhagirath Chauhan, Professor in Weed Science, The University of Queensland

Key words

harvest weed seed control, Queensland, seed bank, weed biology, weed ecology, weed management

GRDC code

UA00156

Take home messages

- Seed persistence (at the surface and 2 cm depth) and seed retention of nine weed species was evaluated
- Surface seeds of feathertop Rhodes grass (FTR) and windmill grass (WMG) persisted for <1 year while common sowthistle and Mexican poppy seeds persisted for <1.5 years. Bladder ketmia and sesbania pea seeds persisted for >3.5 years
- Burial increased seed persistence. Buried seeds of Mexican poppy, turnip weed, button grass, bladder ketmia and sesbania pea persisted for >3.5 years
- Turnip weed plants retained all seeds at wheat harvest, whereas common sowthistle dispersed 95% of seeds. Sesbania pea, FTR, and WMG all had greater than 80% seed retention at mungbean harvest, suggesting the most potential of using harvest weed seed control.

Background

Weeds are one of the most important biological constraints to grain production in Australia. In the northern grain region of Australia, weeds cause an annual revenue loss of $140 million, which includes yield loss in wheat, barley, oats, canola, pulses and sorghum, and yield loss from fallow weeds (Llewellyn et al. 2016). These losses can be reduced by adopting integrated weed management (IWM) programs. However, to develop effective and sustainable IWM programs, information on weed biology is a pre-requisite. For example, knowledge of weed seed bank persistence is critical for the development of IWM tactics for herbicide-resistant or difficult-to-control weeds. However, such information is not available for some major and emerging weed species, especially in the northern grain region.

Similarly, information on weed seed retention at crop harvest may be used to create the opportunity to use harvest weed seed control (HWSC) tactics and prevent the build-up of weed seed banks. However, such information is not available for most weed species in the northern region. In this paper, major findings of the 'Emerging weeds' project are discussed for seed persistence and seed retention of three winter weeds; common sowthistle (*Sonchus oleraceus*), Mexican poppy (*Argemone mexicana*), and turnip weed (*Rapistrum rugosum*) and six summer weeds; grass species: button grass (*Dactyloctenium radulans*), feathertop Rhodes grass (FTR; *Chloris virgata*), liverseed grass (LSG; *Urochloa panicoides*) and windmill grass (WMG; *Chloris truncata*); broadleaf species: bladder ketmia (*Hibiscus tridactylites*) and sesbania pea (*Sesbania cannabina*). Seed retention of winter weeds was assessed in a wheat crop and summer weeds in a mungbean crop.

Materials and methods

Seed collection

Seeds of common sowthistle, Mexican poppy and turnip weed were collected in October/November 2015 from wheat or chickpea fields. Seeds of bladder ketmia, button grass, FTR, LSG, sesbania pea and WMG were collected in March/April 2016 from sorghum, mungbean or summer fallow fields. Seeds of all species were collected from Dalby, Chinchilla or Gatton regions. Seeds were dried in a screenhouse for 2-3 days.

Seed persistence

Fifty seeds of each species were placed (within 7-10 days after collection) in nylon bags and the bags were buried at 2 cm depth or placed on the soil surface in a field at the Gatton campus of the University of Queensland. There were three replications of each treatment. The bags were exhumed at 3, 6, 12, 18, 24, 30, 36 and 42 months after seed placement in the field and seeds were examined in the laboratory for their germination characteristics (germination, dormancy and viability). In this paper, only the viability component (i.e., seed persistence) is presented. Rainfall for the first year after seed placement is shown in Figure 1.

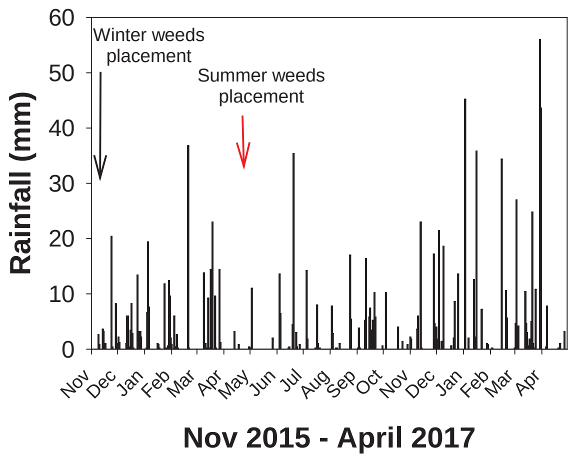


Figure 1. Rainfall (mm) at Gatton from November 2015 to April 2017. Winter weed seeds were placed in the field in November 2015 and summer weed seeds in April 2016.

Seed retention

Winter weeds (common sowthistle, Mexican poppy and turnip weed) were established in a wheat crop. The crop was planted in May 2016 and May 2017 at a seeding rate of 60 kg/ha in 18 cm row spacing using a tyne seeder. Plots were sprinkle irrigated four times on alternate days starting from seeding to ensure good crop and weed emergence. Seed traps (4 trays/plot) were placed in the field to determine the temporal pattern of seed shed in different weed species. Seeds collected in these traps were retrieved carefully from seed maturity until harvest at a weekly interval to quantify the rate of seed shed. For common sowthistle, seed dispersal was computed based on the numbers of flower heads that dispersed or remained on the plant at the time of harvest.

Summer weeds were established in a mungbean crop. The crop was planted in October 2016 and November 2017 at a seeding rate of 30 kg/ha in 50 cm rows using a tyne seeder. After planting, weed seeds were mixed with sand and broadcast. As mentioned above, plots were regularly watered to ensure good crop and weed establishment. The plots were regularly hand-weeded to remove all weeds except the target species. Seed traps, made of PVC pipes and lined with cloth at the bottom to hold seeds and to drain water, were randomly placed in the crop. Seed retention was computed based on the seeds collected in the traps at crop harvest. These results were also confirmed by visually observing the seed head/panicle/inflorescence.

There were three replications of each treatment in each year; however, data for only one year is shown as the response was similar across the two years.

Results and discussion

Seed persistence

Winter weeds

Common sowthistle seeds placed on the soil surface degraded rapidly, with less than 5% of seeds remaining after 12 months (Figure 2). At 18 months after seed placement, persistence of common sowthistle seeds was zero. Buried seeds took 2 years for complete seed depletion. Common sowthistle seeds have no to very low dormancy levels (Manalil et al. 2018), suggesting that most seeds will germinate after good rainfall events and result in the rapid depletion of the seed bank. Seed burial, by creating dark conditions, may enforce dormancy in some seeds, resulting in longer persistence for buried seeds compared with surface seeds.

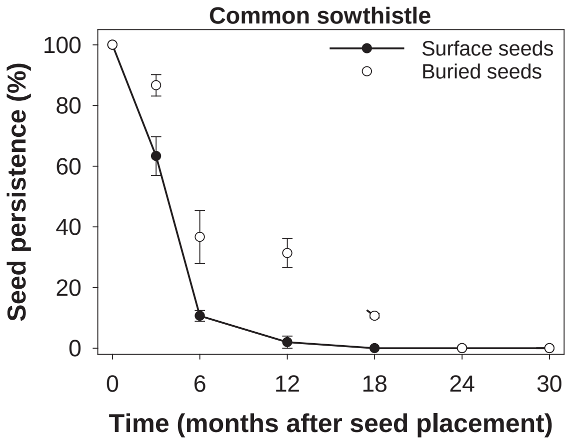


Figure 2. Persistence of common sowthistle seeds placed on the soil surface or buried at 2 cm.

Mexican poppy seeds placed on the soil surface depleted very rapidly, resulting in <10% viable seeds after 3 months (Figure 3). These results of the rapid depletion of the surface seeds were confirmed in another study (data not shown). No seeds remained viable on the soil surface at 18 months. In contrast, this species showed high persistence levels for buried seeds. At 42 months, up to 33% of buried seeds were still viable, suggesting that burial by tillage or planting operations will increase the persistence of Mexican poppy seeds.

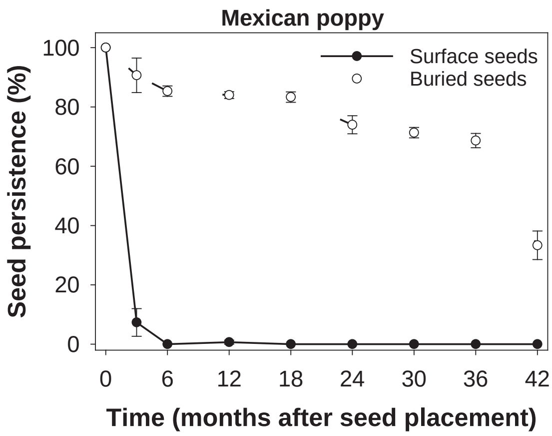


Figure 3. Persistence of Mexican poppy seeds placed on the soil surface or buried at 2 cm.

In 3 months, about 80% of turnip weed seeds placed on the soil surface were depleted (Figure 4). However, it took 30 months to deplete the rest of the seed bank. Seeds of this species are highly dormant, which is mainly due to the pod wall (Chauhan et al., 2006). Seeds on the soil surface experience high fluctuations in temperature and moisture conditions, which soften the pod wall and allow for high seed germination and depletion. Burial of seeds by any means will delay the decay of the pod wall, resulting in longer persistence of buried seed banks. About 8% of buried seeds of turnip weed were still viable at 42 months (last exhumation in this study).

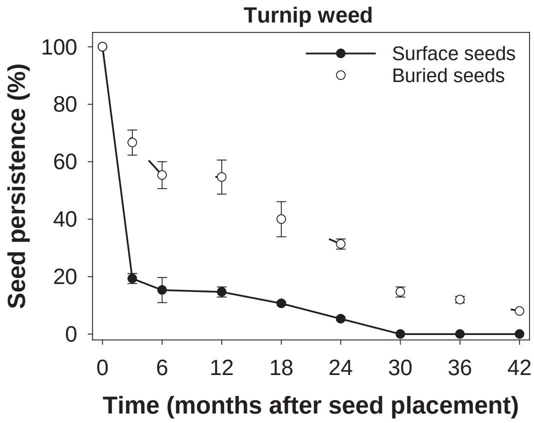


Figure 4. Persistence of turnip weed seeds placed on the soil surface or buried at 2 cm.

Summer weeds

Grass species: About 15% of button grass seeds remained viable on the soil surface at 12 months after placement on the soil surface and 5% remained at 24 months (Figure 5). It took 3 years to completely deplete the seed bank of button grass on the soil surface. Buried seeds persisted much longer; 9% of seeds were viable at 42 months. Physical scarification stimulates seed germination in button grass, suggesting that seed burial will delay the natural scarification process and result in longer persistence of button grass seeds.

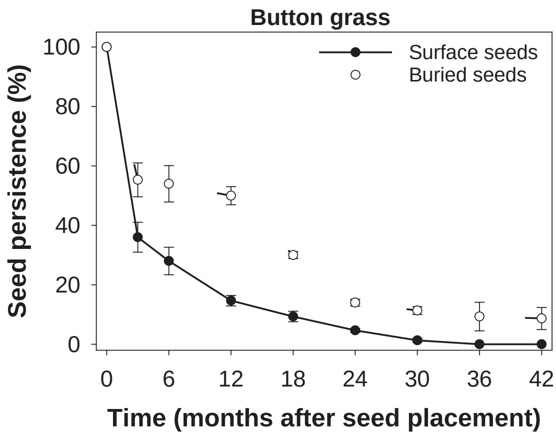


Figure 5. Persistence of button grass seeds placed on the soil surface or buried at 2 cm.

Feathertop Rhodes grass (FTR) seeds on the soil surface depleted very fast (Figure 6). Only 31% of seeds remained viable at 6 months and no surface seeds were found to be viable at 12 months. Buried seeds persisted slightly longer with 9% viable seeds at 12 months and no viable seeds at 18 months. FTR seeds have a moderate level of dormancy and can germinate at a wide range of temperatures ranging from 15/5 °C alternating day/night temperatures to 35/25 °C temperature regimes. Good rainfall events in the first 12 months might have resulted in the germination of most seeds (Figure 1).

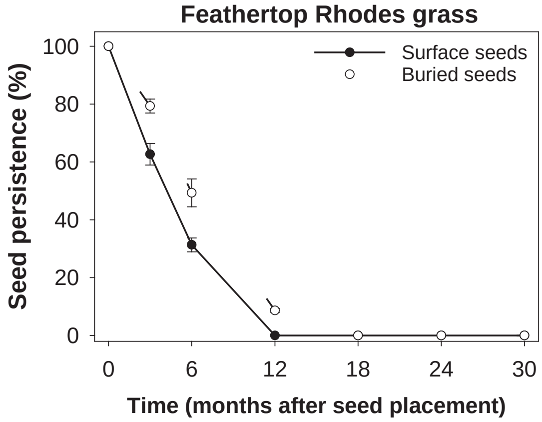


Figure 6. Persistence of feathertop Rhodes grass seeds placed on the soil surface or buried at 2 cm.

The depletion of the liverseed grass (LSG) seed bank on the soil surface was significant with 15% of seeds remaining at 12 months, less than 1% at 18 months and no viable seeds at 24 months after seed placement (Figure 7). Buried seeds persisted longer with 12% viable seeds at 24 months and 3% at 36 months. It took 3.5 years to completely exhaust the buried seed bank of LSG. In our lab studies, LSG was found to have a moderate level of seed dormancy. Fresh seeds had 40% germination, which increased to 90% after 4 months. These results suggest that most LSG seeds will germinate in the first years after shedding but some seeds may remain ungerminated in the soil for another two years.

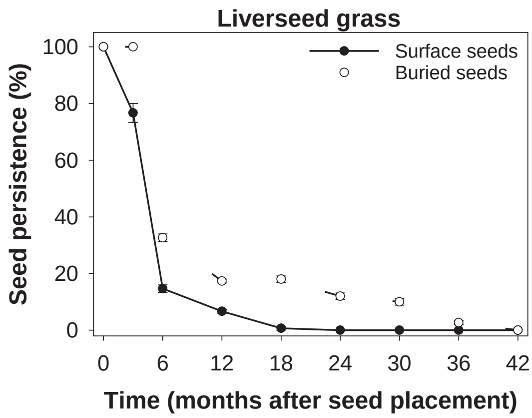


Figure 7. Persistence of liverseed grass seeds placed on the soil surface or buried at 2 cm.

Windmill grass (WMG), a closely related species of FTR, seeds behaved similarly to FTR on the soil surface. Total seed depletion was observed at 12 months after seed placement (Figure 8). Buried seeds of WMG persisted longer than FTR seeds. About 20% of buried seeds were viable at 12 months and 3% at 18 months but no seeds survived at 24 months.

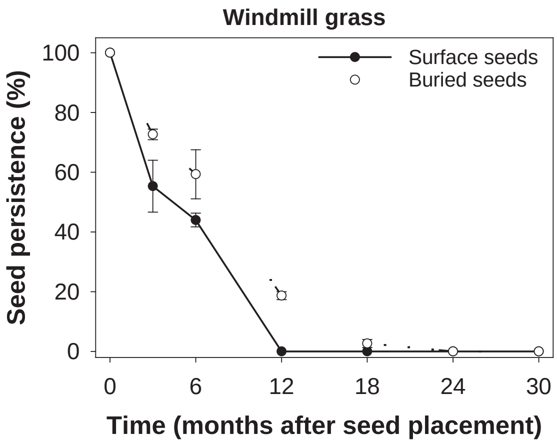


Figure 8. Persistence of windmill grass seeds placed on the soil surface or buried at 2 cm.

Broadleaf species: Among all the studied species, bladder ketmia had the greatest level of seed bank persistence. About 3% of the seeds were still viable at 42 months after placement on the soil surface (Figure 9). At the same time, 63% of seeds were viable at 2 cm depth. The time required to lose half of the life of buried bladder ketmia seeds could not be established in this study. Elsewhere, about 25% of bladder ketmia seeds survived after 7 years of burial (Uremis and Uygur, 2005; Westre et al., 1996). This species has hard-seeded or physical dormancy (Chauhan, 2016), suggesting that seed depletion will be quicker for seeds present on the soil surface than when buried in the soil.

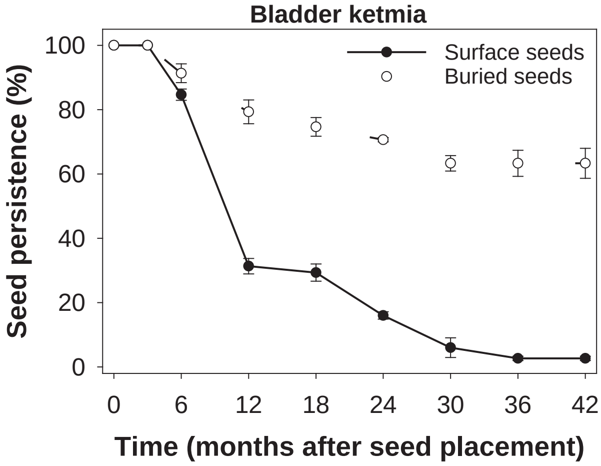


Figure 9. Persistence of bladder ketmia seeds placed on the soil surface or buried at 2 cm.

Sesbania pea behaved similarly to bladder ketmia as both species are known to have physical seed dormancy (Iqbal et al., 2019). However, the seed bank persistence level of sesbania pea was lower than bladder ketmia. About 1% of surface seeds and 8% of buried seeds were viable at 42 months after seed placement (Figure 10).

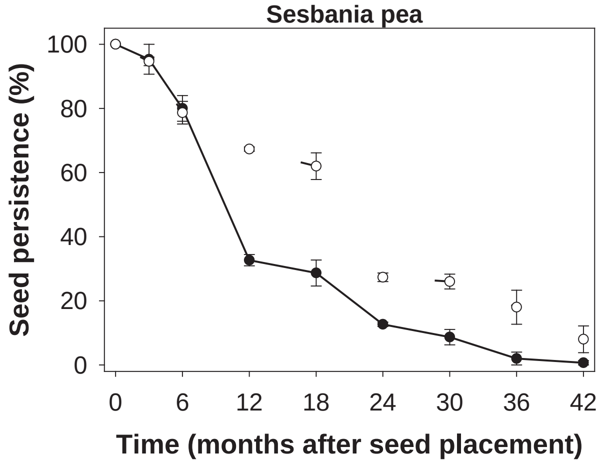


Figure 10. Persistence of seasbania pea seeds placed on the soil surface or buried at 2 cm.

All nine weed species were arranged in ascending order in terms of time required to complete depletion of their seed banks (Figure 11). FTR and WMG surface seeds took the shortest time (12 months) for complete depletion. Common sowthistle and Mexican poppy took 18 months for 100% seed depletion on the soil surface. Surface seeds of bladder ketmia and sesbania pea, both hard-seeded species, were still viable on the soil surface at 42 months, suggesting that these seeds will keep germinating in no-till farming systems beyond 3 years of their shedding. Buried seeds of all weed species persisted longer than surface seeds. This is not a surprising result as buried seeds have reduced risks from seed predators and fatal-germination factors such as high fluctuations in temperature and moisture conditions.

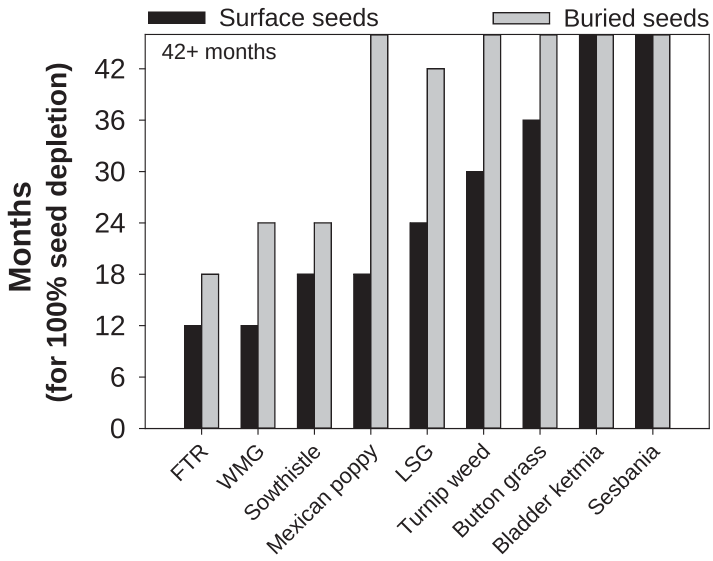


Figure 11. Number of months required to deplete complete seed banks of different weed species. Seeds were placed on the soil surface or buried at 2 cm soil depth. 42+ means more than 42 months.

Seed retention

Winter weeds in wheat

Common sowthistle produced a maximum of 193,000 seeds/m² but 95% of these seeds had dispersed at wheat crop harvest (Figure 12). In contrast, 100% of seeds were retained for turnip weed, which produced 32,000 seeds/m². These results suggest that turnip weed exhibits high seed retention at wheat harvest and employing HWSC practices is likely to reduce its seed bank in the soil (Manalil and Chauhan, 2019). The author observed that turnip weed in a poorly-established crop in the northern grain region can grow more than 4-5 cm in stem diameter which may impede harvest and HWSC suitability. Therefore, in such situations, seed sterilising practices may need to be used.

Seed-capturing practices may not have desired results for common sowthistle in wheat. As common sowthistle matured much earlier than the wheat crop, most seeds had dispersed before crop harvest (Manalil et al., 2020a). These results suggest that a higher level of seeds of this weed species could be captured in a shorter duration crop or variety. Common sowthistle seeds could also be captured from late-emerging weed seedlings.

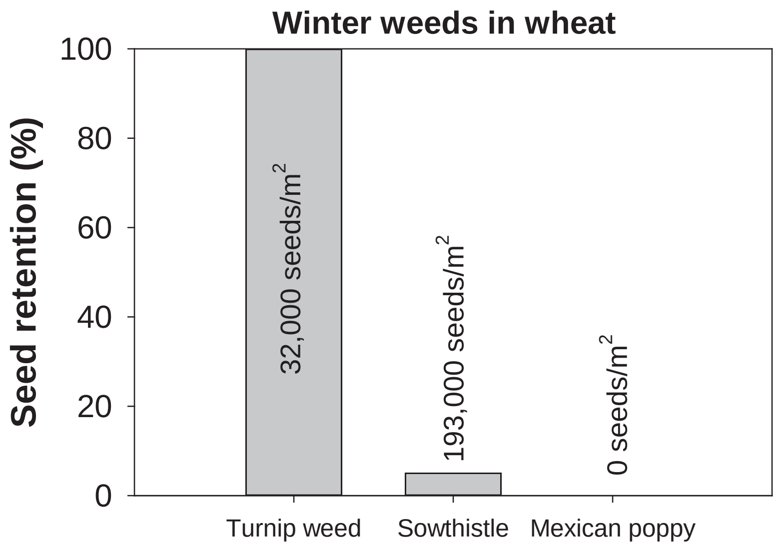


Figure 12. Seed retention (>15 cm from the surface) of winter weeds in a wheat crop. Maximum seed production (seeds/m^2) by these species is also given as text.

Mexican poppy emerged was severely impacted through competition from the 18 cm row wheat crop (Manalil and Chauhan, 2019). These weed plants reduced wheat yield by 20% but did not produce a single seed, suggesting the weed-suppressive benefits of narrow-row planted crops. However growers have observed Mexican poppy seed production in wider row winter cereals, chickpea and winter fallow fields.

Summer weeds in mungbean

Among the broadleaf weed species, sesbania pea retained 100% of seeds at mungbean harvest and bladder ketmia retained 60% of seeds (Figure 13). Among the grasses FTR, WMG, LSG and button grass retained about 95, 85, 75 and 70% of seeds at mungbean harvest. Weeds, such as button grass and FTR, produced more than 100,000 seeds/m², but seed retention of at least 60% suggests opportunities for HWSC. Similar results may not be possible in a longer-duration summer crop, for example, sorghum. In a previous study, only 60% of FTR seeds were retained at sorghum crop harvest (Mahajan et al., 2020).

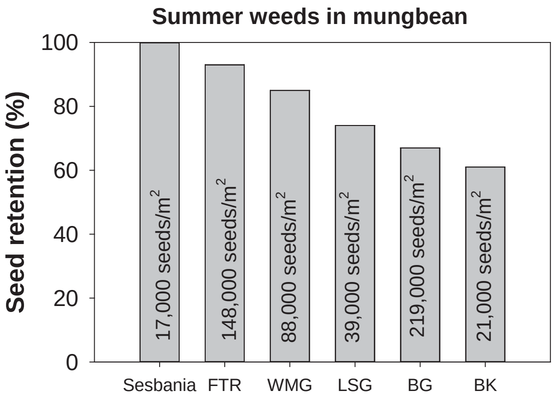


Figure 13. Seed retention of summer weeds in a mungbean crop. Maximum seed production (seeds/m^2) by these species is also given.

Turnip weed, button grass, bladder ketmia and sesbania pea produced a significant number of seeds (Figures 12 and 13), which were found to persist longer than 3.5 years (Figure 11). These characteristics enhance the adaptive potential of such weed species to become problematic and hard-to-control weeds. However, high seed retention and differential maturity periods between crops and weeds provide an opportunity for controlling such weed species through HSWC practices. Crop topping and desiccants may also be possible options in some crops.

FTR is becoming an increasing problem in the northern grain region of Australia. In these trials, it produced almost 150,000 seeds/m² in mungbean; however, its seeds persisted only for a year on the soil surface and 1.5 years in the soil (2 cm depth). These results suggest that prevention of seed set can rapidly deplete seed banks of FTR, and similar weed species. The most effective management of such weeds is to prevent seed set and drive down the existing seed bank through intensive management.

Conclusion

Weed seed bank persistence was variable between species and depths. Seeds persistence ranged from less than 12 months (for example, FTR and WMG) to more than 42 months (for example, turnip weed, Mexican poppy and button grass). Therefore, IWM programs need to consider the variation between weed species. Burial increased seed bank persistence of all nine weed species, suggesting that sowing operations or soil cracks may increase their persistence even in no-till farming systems.

Future research needs to evaluate the seed bank persistence of different weed species in different rainfall regions, soil conditions and crop residue situations. For example, some weeds in southern Australia were found to persist longer in a drier year compared with a wetter year.

With the exception of Mexican poppy in a highly competitive wheat crop in this trial, all the other eight weed species produced a considerable number of seeds and some with very high dormancy levels (e.g., bladder ketmia and sesbania pea). However, except for common sowthistle, all other weed species retained at least 60% of seeds at crop maturity. There is the potential for seed-sterilising and seed-capturing technologies which can reduce further seed input to the soil seed bank. It should be noted that no herbicide weed control was used in this study. Later weed germinations, which often are not controlled by herbicides, may have higher seed retention at harvest. Therefore, there is a need to evaluate seed retention of these weed species in combination with different weed control practices. Similarly, crop planting time and crop row spacing may also affect seed retention by affecting the competitive ability of the crop. Seed retention can also be influenced by weed seedling emergence time through affecting weed phenology, including weed height. Before HWSC practices become a necessary component in the northern region, there is a need to evaluate seed retention affected by the above-said factors.

References

Chauhan BS (2016) Germination biology of *Hibiscus tridactylites* in Australia and the implications for weed management. Scientific Reports 6:26006

Chauhan BS, Gill G, Preston C (2006) Factors affecting turnipweed (*Rapistrum rugosum*) seed germination in southern Australia. Weed Science 54:1032-1036

Iqbal N, Manalil S, Chauhan BS, Adkins SW (2019) Germination biology of sesbania (*Sesbania cannabina*): An emerging weed in the Australian cotton agro-environment. Weed Science 67:68-76

Llewellyn RS, Ronning D, Ouzman J, Walker S, Mayfield A, Clarke M (2016) Impact of weeds on Australian grain production: the cost of weeds to Australian grain growers and the adoption of weed management and tillage practices. Report for GRDC. CSIRO, Australia. In., 112.

Mahajan G, Walsh M, Chauhan BS (2020) Junglerice (*Echinochloa colona*) and feather fingergrass (*Chloris virgata*) seed production and retention at sorghum maturity. Weed Technology 34:272-276

Manalil S, Ali HH, Chauhan BS (2018) Germination ecology of *Sonchus oleraceus* L. in the northern region of Australia. Crop and Pasture Science 69:926-932

Manalil S, Ali HH, Chauhan BS (2020a) Interference of annual sowthistle (*Sonchus oleraceus*) in wheat. Weed Science 68:98-103

Manalil S and Chauhan BS (2019) Interference of turnipweed (*Rapistrum rugosum*) and Mexican pricklepoppy (*Argemone mexicana*) in wheat. Weed Science 67:666-672

Manalil S, Mobli A, Chauhan BS (2020b) Competitiveness of windmill grass (*Chloris truncata*) and feathertop Rhodes grass (*Chloris virgata*) in mungbean (*Vigna radiata*). Crop and Pasture Science 71:916-923

Uremis I and Uygur FN (2005) Seed viability of some weed species after 7 years of burial in the Cukurova region of Turkey. Asian Journal of Plant Sciences 4:1-5

Westra P, Pearson CH, Ristau R, Schweissing F (1996) Venice mallow (*Hibiscus trionum*) seed production and persistence in soil in Colorado. Weed Technology 10:22-28

Acknowledgements

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. The author would also like to thank Sudheesh Manalil for conducting the trial in the initial years of the project.

Contact details

Bhagirath Chauhan (Sunny)
The University of Queensland
Building 8115, Gatton, Queensland 4343
Ph: 0427 923 272
Email: b.chauhan@uq.edu.au

Wheat 2030 - opportunities in a volatile world

Ross Kingwell, AEGIC

Key words

wheat, markets, diversification, exports

Take home message

Over the last 20 years Australian wheat export sales have pivoted away from Middle East markets towards a greater focus on SE Asian markets. Over the next 10 years wheat production in eastern Australia increasingly will continue to focus on markets in SE Asia and more especially within Australia. Limited growth in wheat production in eastern Australia is expected over the next decade, yet domestic demand, mostly in eastern Australia, will increase due to population growth and food preferences that increasingly draw on feed grains. The trend increase in wheat production in eastern Australia will need to solely go to the domestic market over the next decade. Bumper years, however, will cause marked increases in grain exports. In general, domestic market opportunities will be increasingly important for eastern Australian wheat producers.

The global outlook for wheat

Towards 2030 the global outlook for wheat will be affected by major supply and demand influences. On the supply side, the Black Sea region (Russia, Ukraine, Kazakhstan) and eastern Europe, will sustain their emergence as major sources of competitive wheat exports (Figure 1). As a result, Australia, Canada and the USA will see their shares of global wheat exports being squeezed.

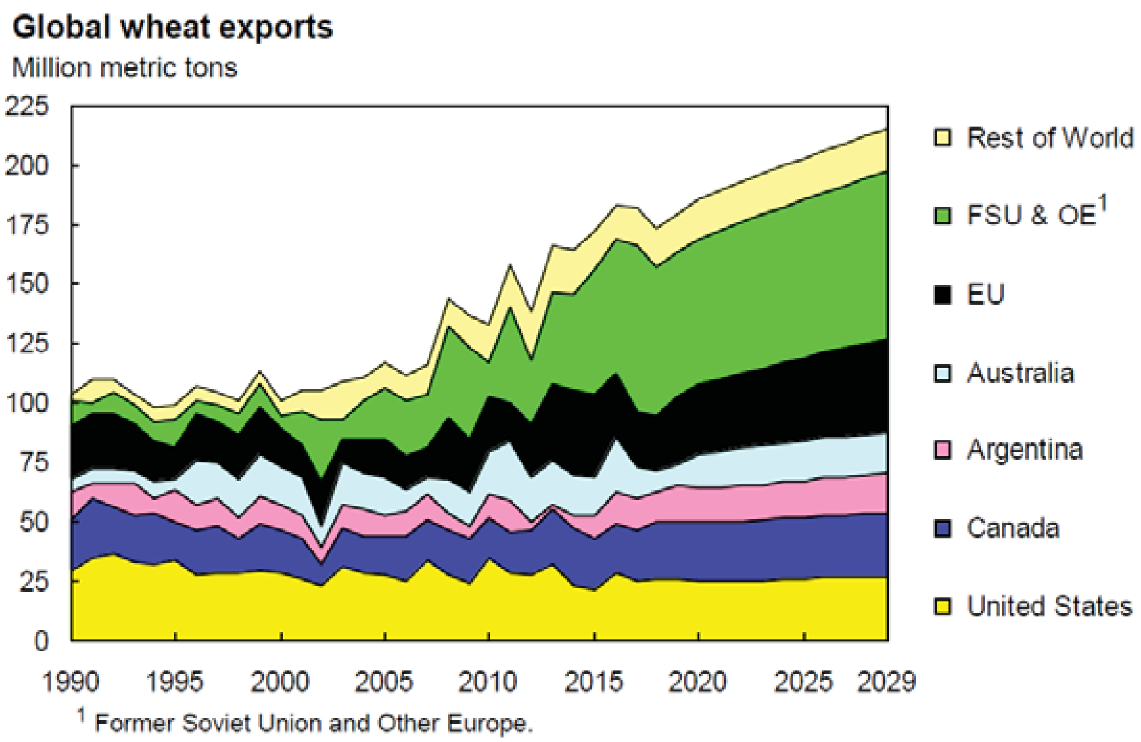


Figure 1. Regional suppliers of wheat exports. Source: USDA (2020)

Wheat exports by Russia and Ukraine are expected to continue their recent pattern of strong export growth and are projected to climb further from 59.9 mmt in 2020/21 to 69.3 mmt in 2029/30. This increase accounts for 31 percent of the projected increase in world wheat exports. EU wheat exports are projected to reach 39 mmt by 2029/30, a 3.4 percent annual growth rate. Rising EU exports are due to higher wheat yields and a decline in the EU use of feed wheat. Argentina's wheat area is expected to grow despite its government applying export taxes on its principal export grains. Argentina's wheat exports are expected to rise to 16.8 mmt in 2029/30, with Brazil continuing to be its principal export destination, projected to import 8.1 mmt by 2029/30.

On the demand side, the global relative importance of wheat markets in SE Asia, the Middle East and Africa will increase (Figure 2). The increase in demand for wheat in these regions principally will be driven by population growth (Figure 3) and to a lesser extent by increases in per capita incomes that will increase the demand for wheat for food and feed purposes.

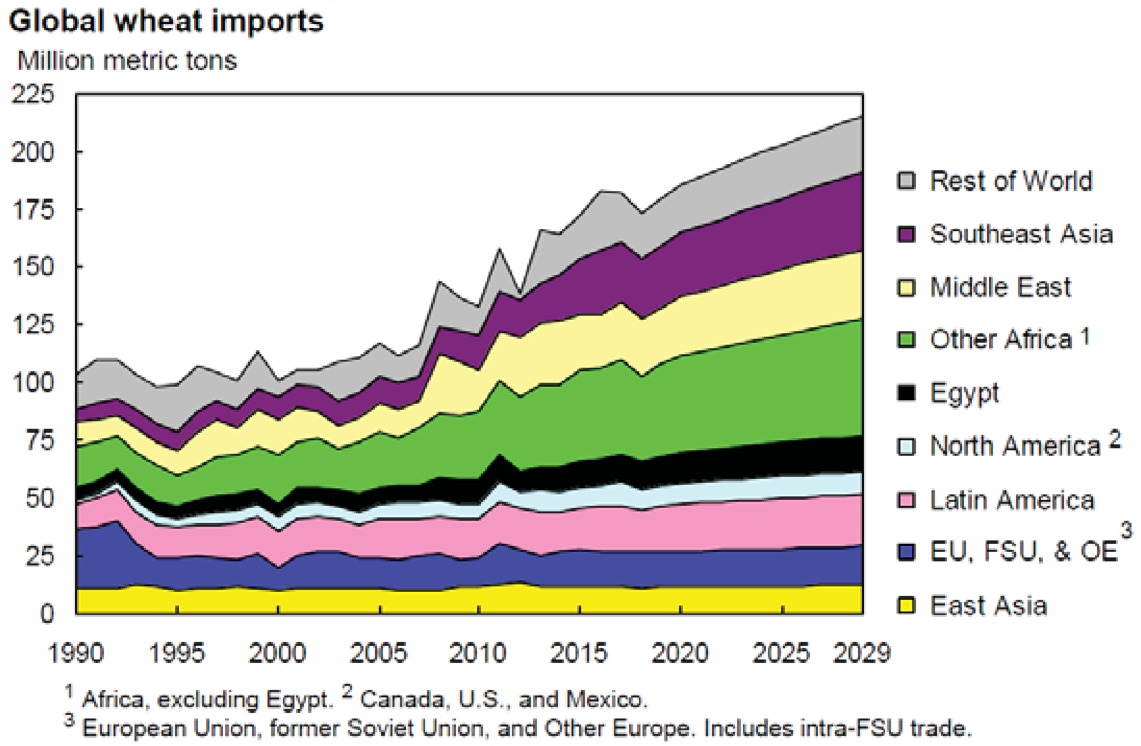


Figure 2. Regional importers of wheat. Source: USDA (2020)

Although China and India together account for over 36 percent of the world's population and might be expected to be major importers of wheat, in fact in most years they import relatively minor or modest amounts due to their large domestic production of wheat. Population growth in both countries is declining (more so in China) with India expected to overtake China as the world's most populous country by 2027.

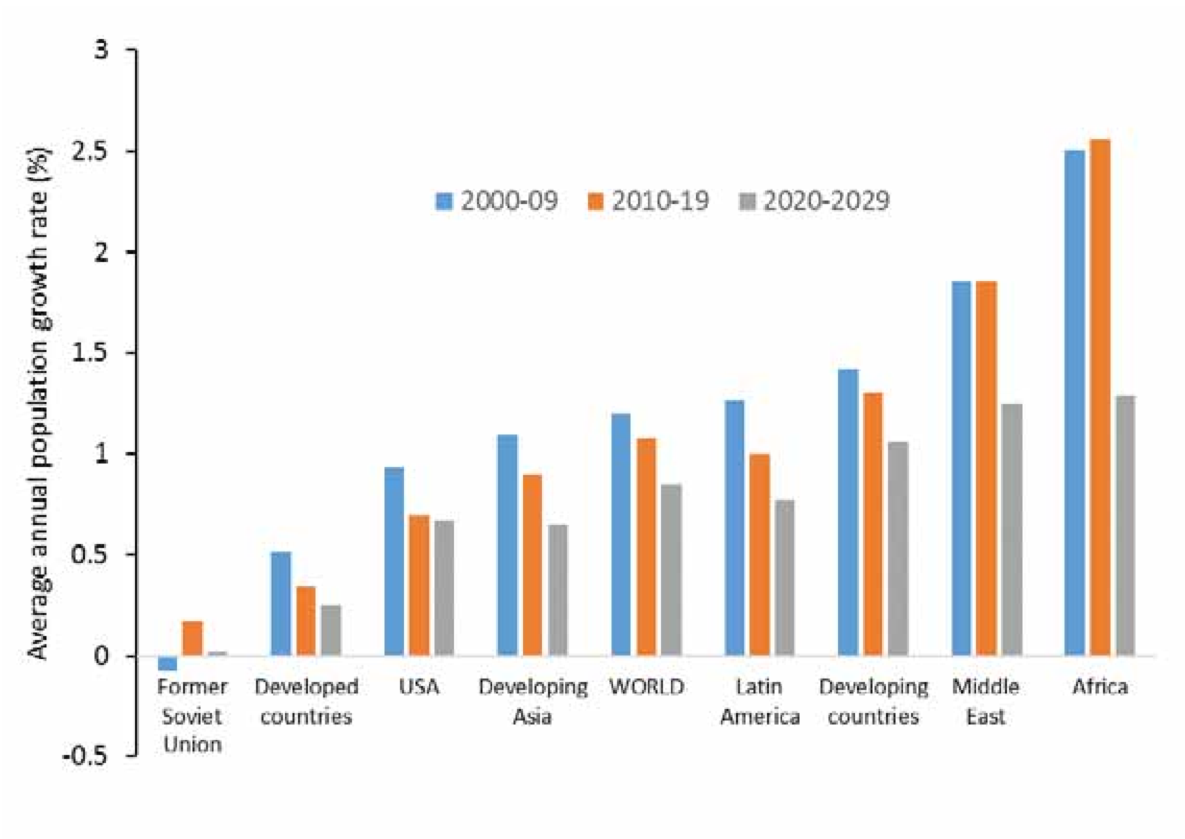


Figure 3. Regional increases in population. Source: Based on USDA (2020)

As shown in Figure 4, apart from Argentina that principally exports its wheat to Brazil, most wheat exporting nations deliver their wheat to a wide range of countries. Some low-cost wheat exporters, such as Russia, Ukraine and Romania, have the added advantage of being able to export to nearby countries such as Egypt, Turkey, Iran and Iraq that, in combination, are major global sources of wheat import demand. Middle East markets that once were key markets for Australian wheat exports are now mostly serviced by cheaper Black Sea country supplies; and so Australia has shifted its wheat export focus towards SE Asia, although the growth in demand for wheat is slowing in SE Asia.

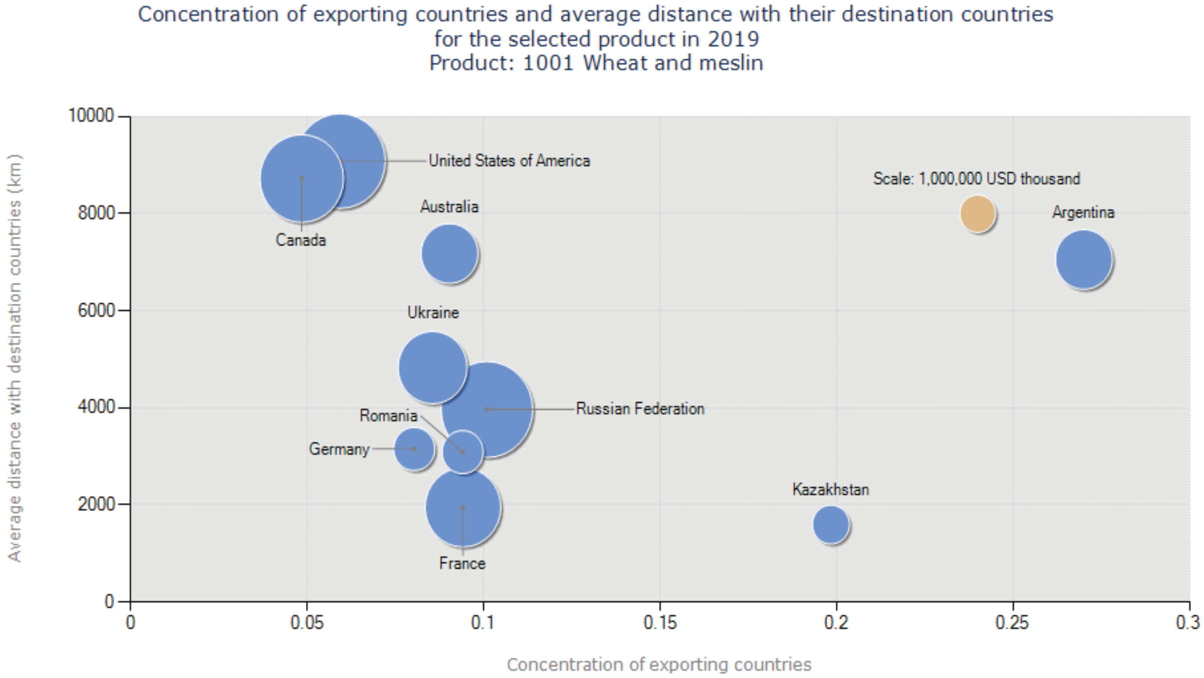


Figure 4. The geographical reach of wheat exported from various main wheat exporting nations. Source ITC (2020)

The domestic market for Australia's wheat

Average wheat production in Australia from 2016 to 2020 was 23.3 mmt. Despite continuing climate volatility, further increases in wheat yields are expected due to improved plant genetics, better agronomic management, soil amelioration and machinery advancements. But the area sown to wheat is not likely to alter much as farmers maintain their crop diversity and rebuild sheep flocks and cattle herds. If the area sown to wheat is maintained at around 12 mha then an additional 2.3 mmt of wheat is likely to be produced in Australia by 2030. However, as shown in Figure 5, consumption of wheat for feed (e.g., poultry, eggs, cattle) is projected to increase, underpinned by a 4.2 million growth in Australia's population. As a result, all of the additional production of wheat in eastern Australia towards 2030 will need to flow to the east coast domestic market, as that is where most of the increased demand for wheat will arise. Consequently, the domestic market will become increasingly important to eastern Australian wheat producers, especially in years when wheat production is constrained by seasonal conditions.

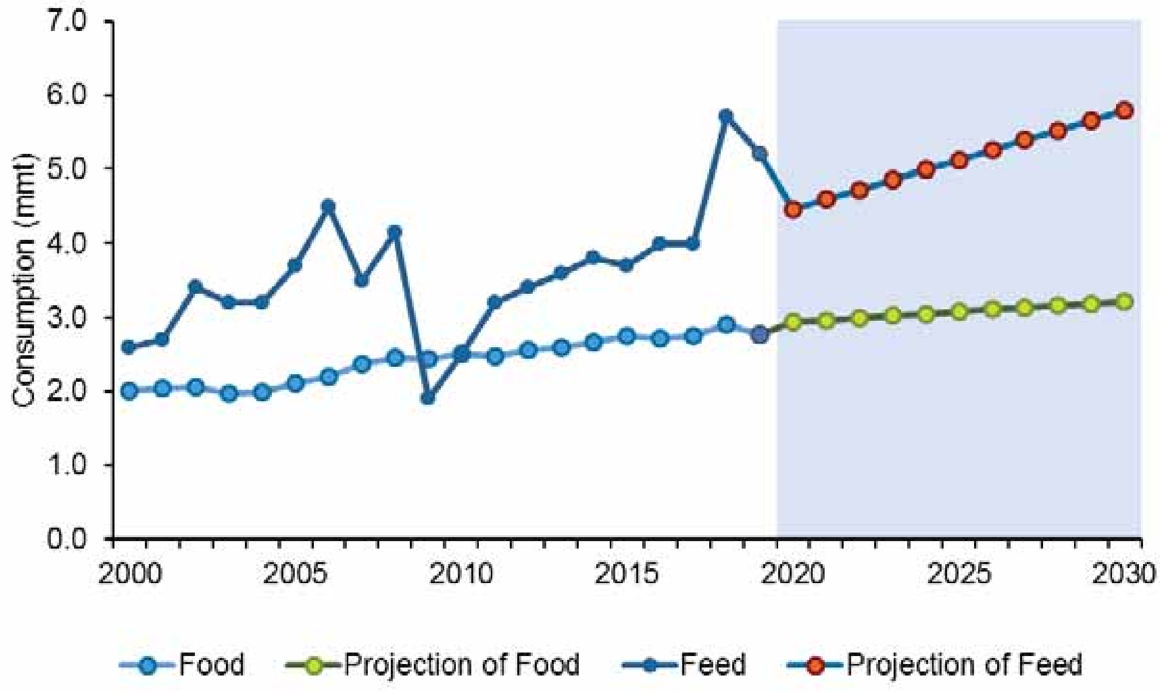


Figure 5. Domestic consumption of wheat as food and feed (including industrial uses). Source: AEGIC based on USDA projections

Transport infrastructure developments such as the inland rail, towards 2030, will facilitate the regional flow of wheat to serve domestic needs of rural industries and major population centres. In dry years when grain production is limited, supplementary grain feeding will increase, creating a spike in demand for feed grains and a reduction in wheat exports (Figure 5 – 2000s and 2018/19).

The importance of the domestic market for wheat, particularly in eastern Australia, is highlighted in Table 1. The likelihood towards 2030 is that growth in wheat exports from Australia's eastern states will be constrained by their increased domestic demand for wheat for feed and food purposes. By contrast, due to their much smaller populations and more reliable grain producing environments, WA and SA will remain main sources of wheat exports. By contrast, due their combination of volatile production, growing domestic markets and a constrained outlook for additional wheat production, states like Qld and NSW will remain less reliable and more opportunistic in their export of wheat.

Table 1. Wheat production and exports from each Australian State (average of 2015 to 2019)

Wheat item	NSW	Qld	Vic	SA	WA
Production (mmt)	5.07	0.88	3.21	4.02	8.33
Exports (mmt)	1.45	0.33	1.61	3.22	7.48
Percent exported (%)	29	37	50	80	90
Coefficient of variation of exports (%)	52	42	46	19	18

Source: ABS

For many wheat importing countries, especially in SE Asia, Australia's small population, relative to its wheat production, combined with its political stability, allows Australia to be a nearby, often reliable, supplier of wheat. By contrast some other wheat exporting nations display political and economic instability that weakens those countries' reliability as wheat exporters. The policy reactions of imposing wheat export taxes or restrictions on wheat exports, as observed in Argentina and Russia, ultimately work to Australia's favour. Furthermore, Australia's wide spread of wheat export markets makes Australian wheat producers, unlike its barley, lobster or wine producers, less exposed to the changeable import policies of any one market.

Contact details

Professor Ross Kingwell
Australian Export Grains Innovation Centre
Perth & Sydney
Ph: 08 6168 9920
Email: ross.kingwell@aegic.org.au

Phenology is FUNdamental – applying key project learnings to optimise grain yield

Felicity Harris[1], Hugh Kanaley[1], Rick Graham[1], Stephen Morphett[1], Peter Matthews[1], Peter Roberts[1], David Burch[1], Nick Moody[1], Greg Brooke[1], Hongtao Xing[1] and Michael Mumford[2]

[1] NSW Department of Primary Industries
[2] Department of Agriculture and Fisheries, Queensland

Keywords

optimal flowering period, frost, heat stress, risk

GRDC code

DAN00213 (Grains Agronomy and Pathology Partnership, GRDC and NSW DPI)

Take home messages

- Aim to target the optimal flowering period (OFP) for your growing environment – know your risk
- Match varietal phenology and sowing time – use the right variety for the right sowing date
- Remain disciplined with sowing dates – don't sow too early!

Background

The 'Optimising yield potential of winter cereals in the Northern Grains Region' project has been a co-investment by GRDC and NSW DPI under the Grains Agronomy and Pathology Partnership (GAPP) in collaboration with QDAF. From 2017-2020, field experiments were sown across ten locations in New South Wales and Queensland where annual rainfall ranged from 184 to 853 mm and grain yields ranged from 0.2-10 t/ha. The project aimed to provide a better understanding of wheat phenology and yield responses across different environments to refine sowing recommendations and improve yield stability and profitability of growers in the northern grains region (NGR).

The consistent messages presented from the project to date have centered around synchronising crop development (phenology) with seasonal conditions to ensure that the optimal flowering period (OFP) is matched to the growing environment. In previous GRDC Update papers, we combined model simulation and field validation to determine OFPs across locations in the NGR, whereby the OFP varied in timing and duration, as well as for different yield levels (Harris *et al.* 2019, 2020). As flowering time is a function of the interaction between variety, management and environment, the two primary management levers growers can pull to ensure flowering occurs at the optimal time and yield is maximised are *time of sowing* and *variety selection*.

Apply these key project findings to reduce risk and optimise grain yield

Understand differences in phenology of commercial wheat varieties

In wheat, flowering is generally accelerated under long-day photoperiods and varieties can be broadly classified into two main types: *winter* or *spring*, distinguished by their response to vernalisation and their adaptation to different sowing dates. Winter types can be sown early and will remain vegetative for an extended period (e.g. DS Bennett⚅, LRPB Kittyhawk⚅). This extended vegetative period arises from a genetic requirement of the plant to experience prolonged cold temperatures to trigger flowering (vernalisation). This delays cold-sensitive reproductive stages to ensure flowering coincides with optimal seasonal conditions in spring.

In contrast, spring types generally do not require a period of cold temperature to initiate flowering. However, they can vary in their responses whereby exposure to cold temperatures can hasten their development (e.g. LRPB Nighthawk⊕) or have no effect on development (e.g. Dart⊕, Sunprime⊕). There are multiple combinations of vernalisation and photoperiod genes which influence phenology responses, as such we have observed significant variation in flowering response among both winter and spring types across environments and in response to sowing time. The figures below present the range in flowering date responses across years and sowing dates for two sites and highlights the risk of precocious flowering in quicker spring types.

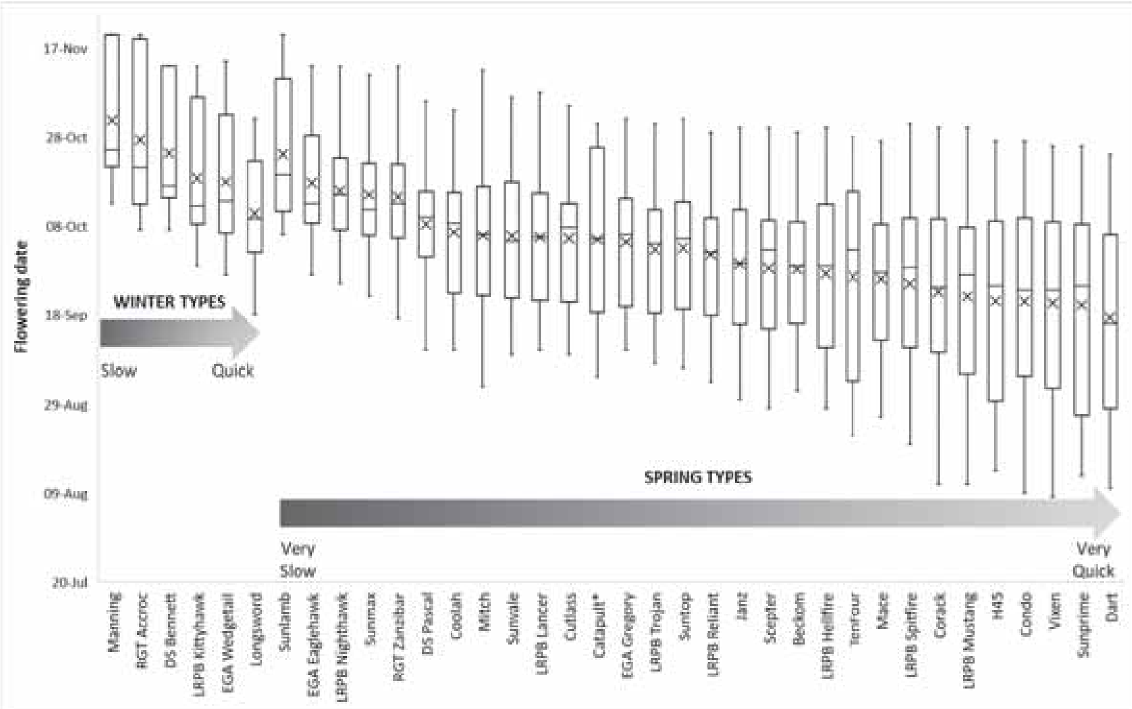


Figure 1. Flowering date responses of genotypes across four sowing dates from early-April to late-May for Wagga Wagga (2018), Marrar (2019) and Harefield (2020) experiments. Asterisk indicates Catapult⊕ only evaluated in 2019, 2020 seasons.

(Varieties Manning, Bennett, Kittyhawk, Wedgetail, Longsword, Sunlamb, Nighthawk, Sunmax, Zanzibar, Pascal, Coolah, Mitch, Lancer, Cutlass, Catapult, Gregory, Trojan, Suntop, Reliant, Scepter, Beckom, Hellfire, TenFour, Mace, Spitfire, Corack, Mustang, Condo, Vixen, Sunprime and Dart are varieties protected under the Plant Breeders Rights Act 1994.)

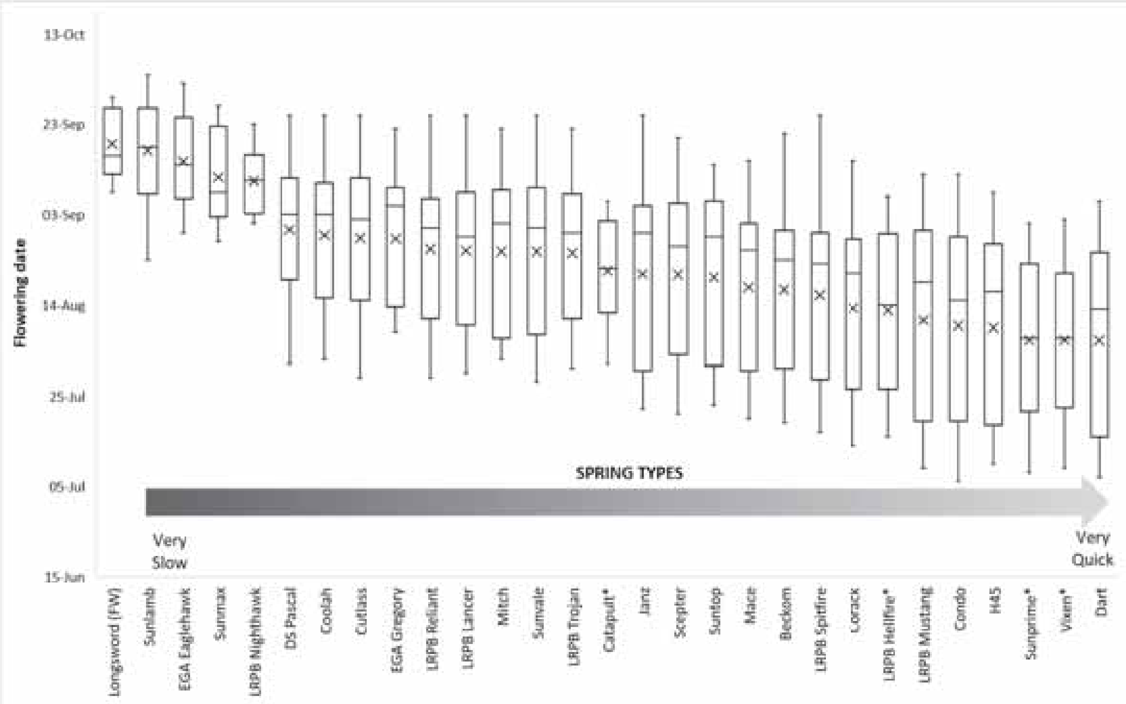


Figure 2. Flowering date responses of genotypes across three sowing dates from mid-April to late-May for Edgeroi (2017) and Narrabri (2019, 2020) experiments. FW – Fast winter type, asterisk indicates genotype only evaluated in two years at Narrabri (2019, 2020).

(Varieties Longsword, Sunlamb, Sunmax, Nighthawk, Pascal, Coolah Cutlass, Gregory, Reliant, Lancer, Mitch, Trojan, Catapult, Scepter, Suntop, Mace, Beckom, Spitfire, Corack, Hellfire, Mustang, Condo, Sunprime, Vixen and Dart are varieties protected under the Plant Breeders Rights Act 1994.)

Need to match varietal phenology and sowing time with environment

Generally, slower developing winter types are more suited to cooler, medium-high rainfall environments in southern NSW which have a longer growing season and increased risk of frost. Winter types when sown early are capable of high water-limited yields, can spread sowing risk and also be a useful frost mitigation tool. However, the vernalisation requirement of these winter types makes them less suited to warmer environments of northern NSW and QLD, where we observed flowering dates later than the OFP and often significant yield penalties in comparison to more adapted mid-quick spring types sown within their recommended window.

In contrast, quicker developing spring types are better adapted to regions with shorter growing seasons, and in environments or later sowing scenarios where frost and heat stress occur in close proximity to each other. Despite the variability across environments and seasons, we were able to identify varieties which were able to maintain relatively stable grain yields across many sowing dates at some sites and which may offer more flexibility to growers.

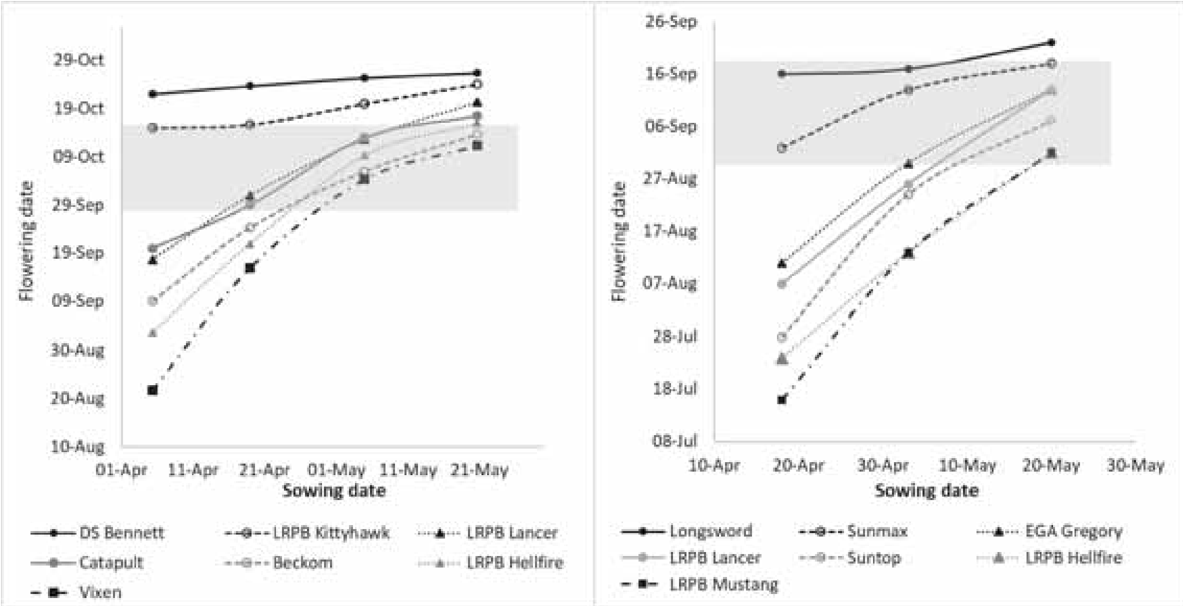


Figure 3. Mean flowering date responses of selected genotypes at a) Wagga Wagga (2018), Marrar (2019) and Harefield (2020) and b) Edgeroi (2017) and Narrabri (2019, 2020) across sowing dates. Shaded box indicates optimal flowering period for each location.
(All the varieties mentioned in the figure above are varieties protected under the Plant Breeders Rights Act 1994.)

Know the risk of getting it wrong - #SowSlowEarly

Winter types – we have observed a yield penalty in many mid-fast winter types when sown prior to early April. This is often due to difficulties establishing crops when soil temperatures are high and there is rapid drying of the seed bed following a rain event. The vernalisation response of some winter varieties has also been observed to be less stable when sown very early, which can result in stem elongation occurring earlier and an increased risk of frost damage. Unless grazing is the primary purpose, sowing prior to early April can increase crop water use under warm temperatures. This can lead to increased likelihood of excessive vegetative growth, taller crop heights and lodging, as well as increased disease pressure.

Spring types – although we have observed significant variation in phenology responses to sowing date amongst spring types, when sown early (when temperatures are warmer and days longer) flowering behaviour is unpredictable and varies substantially across seasons (Figure 1, Figure 2). As such there is increased risk of spring types flowering earlier than the OFP and grain yield penalties due to frost damage or insufficient accumulation of biomass when sown earlier than their recommended windows.

Key recommendation is to remain disciplined with sowing dates – don't sow too early!

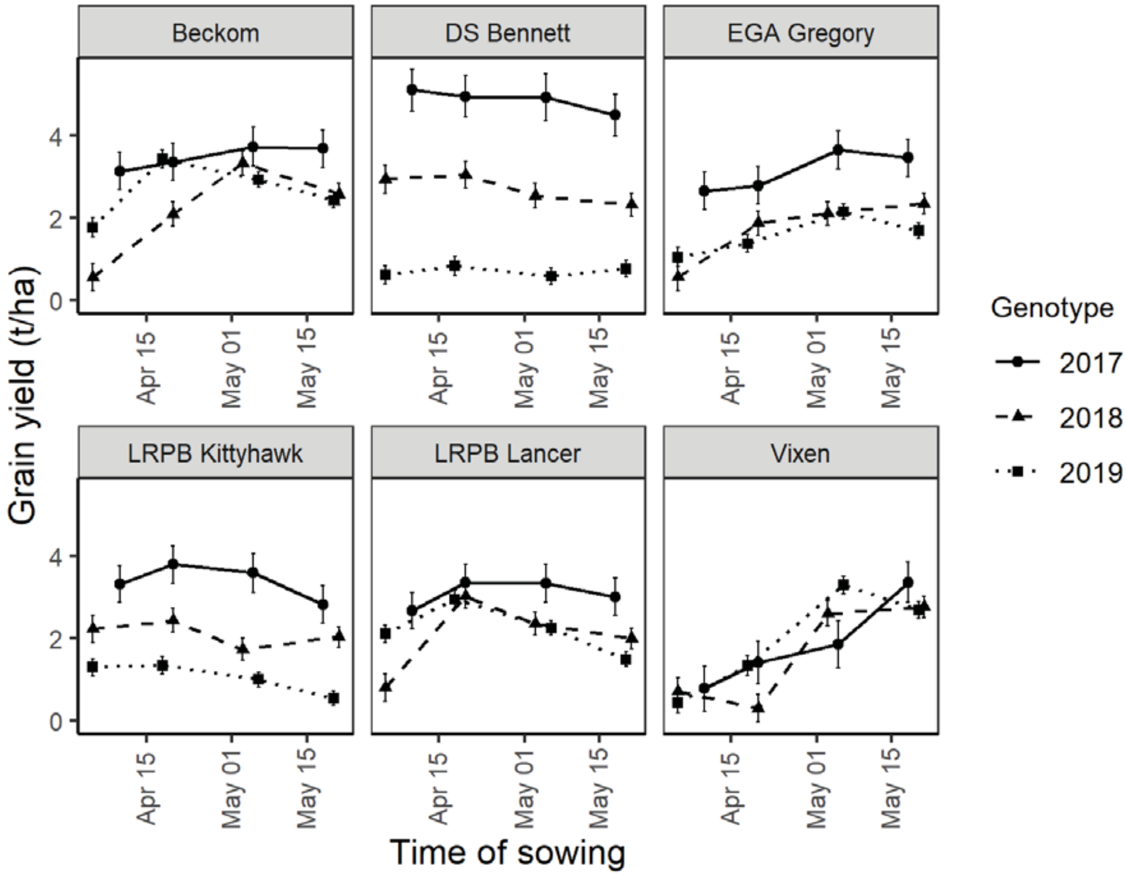


Figure 4. Yield response to sowing time predictions (BLUPs) for a subset of six genotypes for Wagga Wagga in 2017 and 2018 and Marrar in 2019. Vertical error bars denote the 95% confidence interval.
(All the varieties mentioned in the figure above are varieties protected under the Plant Breeders Rights Act 1994.)

Comparing wheat and barley - watch this space...

Compared to wheat, barley is considered to be more widely adapted, have superior frost tolerance and offer higher yield potential across environments in southern Australia. A comparative analysis of the best performing barley and wheat genotypes (defined as the highest yielding treatment) where experiments were co-located across production environments at eight sites in NSW from 2015-2020 is presented in Figure 5. The OFP where yield was maximised for barley was found to be 10-14 days earlier than for wheat, and barley maintained a yield advantage over wheat at yield levels ranging from 1.5-9.6 t/ha. Our understanding of the specific interactions between phenology and environment in barley and the differences in yield physiology between the cereals species is limited. In 2020, a new project was initiated to quantify the flowering time, yield and quality responses of barley, relative to wheat across a range of sowing times. *Preliminary data from 2020 will be presented at the updates.*

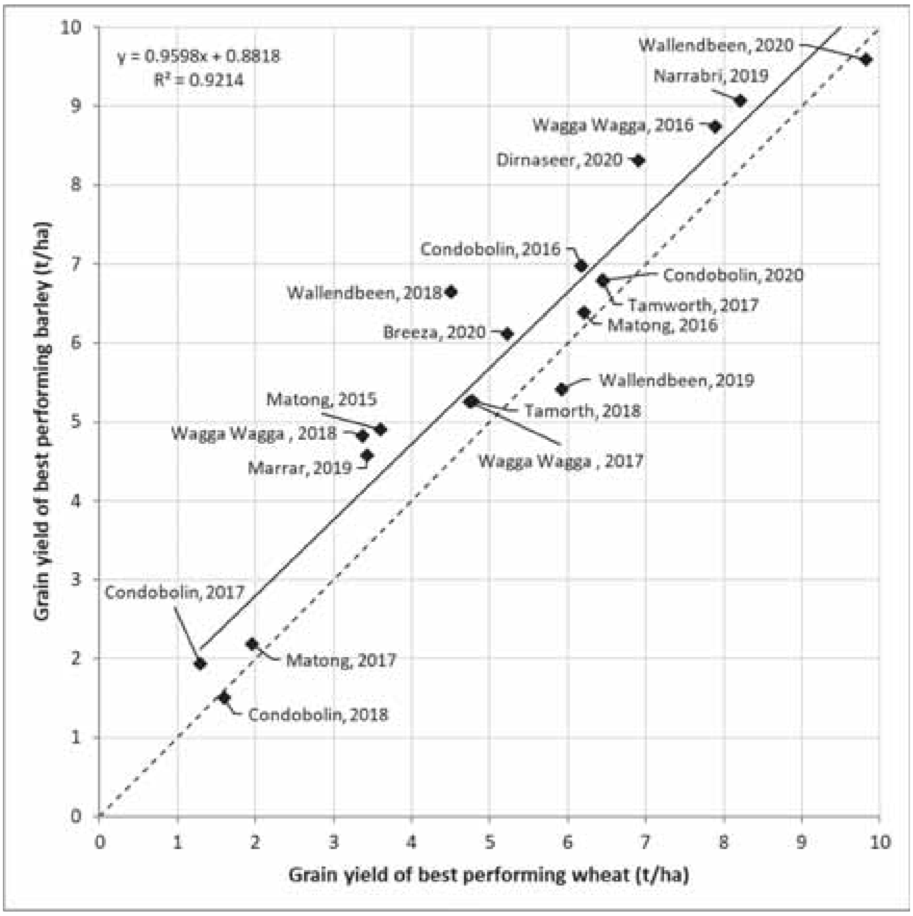


Figure 5. The relationship between the best performing wheat and barley genotypes (sown at optimal time for each environment from mid-April to late-May) at nine sites in NSW from 2015-20. Dotted line indicates 1:1 relationship.

References

Harris, F, Xing, H, Brooke, G, Aisthorpe, D, Matthews, P, Graham, R (2020) Yield stability across sowing dates: how to pick a winner in variable seasons? Proceedings of the GRDC Grains Research Updates, presented at Wagga Wagga, Corowa, Dubbo, Nyngan, Goondiwindi, Barellan and Lake Cargelligo.

Harris, F, Graham, R, Burch, D, Brooke, G, Matthews, P, Xing, H (2019) Hot vs Cold: balancing the risks associated with abiotic stresses in wheat. Proceedings of the GRDC Grains Research Updates, presented at Dubbo and Spring Plains, 2019.

Useful resources

https://grdc.com.au/ten-tips-for-early-sown-wheat

Acknowledgements

Sincere thank you for the technical assistance of Mary Matthews, Jessica Simpson, Ian Menz, Jordan Bathgate and Dean Turner at the Harefield, Dirnaseer and Wallendbeen sites; Braden Donnelly and Daryl Reardon at the Condobolin site and Michael Dal Santo, Jim Perfrement, Jan Hosking and Bruce Haig at the and Narrabri sites in 2020. Thank you also to the Moloney, Hazlett and Ingold families for their support and co-operation in hosting field experiments in 2020.

This research was a co-investment by GRDC and NSW DPI under the Grains Agronomy and Pathology Partnership (GAPP) project in collaboration with Department of Agriculture and Fisheries, Queensland. We would specifically like to acknowledge the collaboration with Darren Aisthorpe, Ellie McCosker and Jane Auer (Department of Agriculture and Fisheries, Emerald), David Lawrence and John Lehane (Department of Agriculture and Fisheries, Toowoomba).

We also acknowledge the input and contribution of James Hunt (La Trobe University) and Kenton Porker (SARDI) throughout the 'Optimising grain yield potential of winter cereals in the Northern Grains Region' project and research collaboration in the GRDC project ULA9175069 'Development of Crop Management Packages for Early Sown, Slow Developing Wheats in the Southern Region.

Contact details

Felicity Harris
NSW Department of Primary Industries, Wagga Wagga
Ph: 0458 243 350
Email: felicity.harris@dpi.nsw.gov.au
@NSWDPI_Agronomy @FelicityHarris6

Rick Graham
NSW Department of Primary Industries, Tamworth
Ph: 0428 264 971
Email: rick.graham@dpi.nsw.gov.au
@NSWDPI_Agronomy @RickGraham15

⯑ Varieties displaying this symbol beside them are protected under the Plant Breeders Rights Act 1994.

Response of wild oats to environmental stresses

Mohammad Ali[1], Alwyn Williams[1], Michael Widderick[2], Steve Adkins[1]

[1] The University of Queensland, School of Agriculture and Food Sciences, Gatton, QLD 4343
[2] Department of Agriculture and Fisheries, 13 Holberton Street, Toowoomba, QLD 4350

Key words

heat stress, drought, no/minimum tillage conservation agriculture, wild oats survival

GRDC code

9175899

Take home messages

- When under heat and water stress, wild oat plants can mature 3 weeks earlier than non-stressed plants
- Stressed plants produce 30% fewer seeds; additionally, these seeds are 40% smaller and show less dormancy than seeds from non-stressed plants
- Seed dormancy can differ between biotypes and between the two types of seeds (primary and secondary) produced by wild oat plants
- Early plant maturation can cause early seed shedding depending on the severity of heat and water stress
- If the occurrence of late season environmental stresses becomes more frequent as forecast under a changing climate, the efficacy of strategies such as harvest weed seed control may further diminish for wild oats.

Background

Wild oats (*Avena fatua* L. and *Avena sterilis* (L.) *ludoviciana* (Durieu) Nyman) are one of the most economically important weeds within the Northern Grains Region (NGR) (Llewellyn *et al*., 2016). In the NGR, ~0.6 M ha of cropping land is infested by wild oats that cost growers ~$4.5 M annual revenue loss by sacrificing crop yield (Llewellyn *et al*., 2016). Wild oats possess a range of survival mechanisms to succeed in the cropping environment, such as variable seed dormancy and a persistent seed bank. In response to changing climatic conditions, where the frequency of hot and dry periods is increasing during the late winter/early spring period (CSIRO, 2011), wild oats plants can mature and shed a majority of their seeds early and before the cereal crop is harvested. These seeds can undergo rapid germination from exposure to cool, wet conditions in the following autumn/winter planting season.

The response of *A. fatua* to a range of environmental stressors has been studied a great deal. However, little research has been conducted on *A. sterilis* ssp. *ludoviciana*, which is the more abundant wild oats species within the NGR (Nugent *et al.,* 1999). The survival mechanisms of *A. sterilis* ssp. *ludoviciana* may differ between biotypes, as has been reported for *A. fatua* (Adkins *et al.,* 1987). Even the primary (the bigger sized seed of a floret) and secondary seeds of *A. sterilis* ssp. *ludoviciana* produced from the same floret may show differences in dormancy behaviour (Quail and Carter, 1968), which may help wild oats to persist in the cropping environment. This study investigated the phenology and reproductive biology of *A. sterilis* ssp. *ludoviciana* biotypes in response to environmental conditions. Specifically, we investigated the impact of heat and water stress, and a combination of both stressors, to gain insight into the potential persistence and invasiveness of wild oats within the NGR.

Objective

To determine how late season heat and water stress, individually and in combination, affect the growth and reproductive biology of *A. sterilis* ssp. *ludoviciana* when applied during the seed development stage – knowledge of which is critical to understand their persistence in NGR cropping systems under changing climatic conditions.

Materials and methods

Seeds of four biotypes were collected from four locations across the NGR: Biloela 1 (-24.35471 °S, 150.49773 °E); Biloela 2 (-24.35048 °S, 150.49767 °E); Toobeah (-28.36792 °S, 149.52197 °E) and Jandowae (-26.66727 °S, 151.02460 °E). The experiment was conducted during June to December 2019, using a completely randomized design with six replications. Three germinated seeds of each biotype were transplanted into a 20 cm diameter × 19 cm height pots each containing 4.5 kg of a black Vertosol soil (71% clay, pH 7.3) obtained from Gatton, QLD. The 96 pots (four biotypes × four treatments × six replications) were maintained at 100% gravimetric plant available water content (PAWC) and kept in a greenhouse under ambient conditions (average ~23/14°C day/night). The stress treatments were applied at panicle emergence (Biloela 1 and Biloela 2 panicle emerged at 58 days, Toobeah 63 days and Jandowae 65 days after seed germination) and were maintained until harvest. The treatments were as per Table 1 below.

Table 1. Stress treatments applied to wild oat populations

	Abbreviation	Treatment
a)	HS	Heat stress at the panicle emergence stage. Pots were moved to a temperature-controlled glasshouse set at 29/23°C 12/12-hour day/night photoperiod and maintained at 100% gravimetric PAWC.
b)	WS	Soil water stress at the panicle emergence stage. Pots were maintained at 60% gravimetric PAWC under the ambient greenhouse conditions.
c)	HWS	A combination of HS and WS. Pots were moved to the same temperature-controlled glasshouse (as mentioned in HS) and maintained at 60% gravimetric PAWC.
d)	Control	No imposed stress. Pots were maintained at 100% PAWC under the ambient greenhouse conditions.

The PAWC of pots was determined by the following equation:

100% PAWC (g H_2O/kg soil) = (water content at field capacity – water content at permanent wilting point) / dry soil weight

The field capacity and permanent wilting point of black Vertosol Gatton soil were determined as 390 and 224 g H_2O/kg soil, respectively. Soil water content of the pots was maintained according to the weight of the pot and considering the weight of the plant.

Data acquisition

The number of days taken to reach maturity, total number of filled primary seeds/plant and filled secondary seeds/plant were recorded for all plants. The 1000 primary and secondary seed weight were determined by taking five lots of 50 seeds from the bulked seed lots (combined across replicates of each stress × biotype treatment) and dried at 80°C for 96 hours. Once dry, they were weighed and values multiplied by four to reach 1000. To check the dormancy status of the freshly harvested hulled primary and secondary seeds, three replicates of 20 filled primary and secondary seeds coming from the bulked seed lots of each treatment were incubated at 9°C under 12/12 hour light/dark condition for 42 days in a germination incubator. Number of seeds germinated was recorded.

Statistical analysis

A. sterilis ssp. *ludoviciana* responses to the different environmental stress treatments were analysed by ANOVA performed in Minitab v. 8.1. Means were separated using Fisher's protected LSD test at $P<0.05$. The graphs were prepared using SigmaPlot v. 13.0.

Results and discussion

Significant two-way interactions between *A. sterilis* ssp. *ludoviciana* biotype and different environmental stressors were observed for all variables studied except for days to maturity (Table 2). Northern NGR biotypes (Biloela 1 and 2) matured in ~97 days, 6 to 8 days earlier than southern biotypes (Jandowae and Toobeah), which matured in ~105 days. However, the stress treatments had a greater impact on driving earlier maturation when compared to the control plants. The HWS treatment had much greater impact on phenology and reproductive biology of *A. sterilis* ssp. *ludoviciana* than when each stressor was applied individually. The HWS treatment resulted in plants maturing 19 days earlier than plants grown without any stress, followed by HS (17 days earlier) and WS (9 days earlier). The exposure of plants to different environmental stressors during the seed development stage stimulated plants to complete their life cycle in a much shorter time. Additionally, exposure to the environmental stressors was responsible for plants producing fewer and smaller seeds as compared to the control plants (Table 2). Primary seed production of plants was reduced by 32, 29 and 12% as compared to the control plants from the HWS, HS and WS treatments, respectively. These figures were similar for the secondary seeds, although it was observed that secondary seed production was less than primary seed production in all biotypes (Table 2). Primary and secondary seed sizes were greatly impacted by the stress treatments. Under HWS, seed size was reduced by as much as ~40% as compared to seeds produced by control plants.

Environmental stressors resulted in a reduction in the dormancy status of the wild oat seeds. The dormancy status of primary seeds produced under stress conditions was reduced from 80, 85, 90 and 87% in the control plants to 23, 45, 62 and 47% in Biloela 1, Biloela 2, Toobeah and Jandowae, respectively, under the combined HWS (Figure 1). The dormancy status of secondary seeds remained higher as compared to the primary seeds; however, HWS produced the least dormant seeds among all treatments for both seed types.

Table 2. The effect of heat and soil water stress treatments applied at panicle emergence on the time of plant maturity, the production of filled primary and secondary seeds, and their seed weight for four *Avena sterilis* ssp. *ludoviciana* biotypes collected from the NGR. Letters within biotypes indicate significant differences among the stress treatments. Main effect means for all biotypes and stress treatments (in bold text) sharing the same letter do not differ significantly. Values in parenthesis are those when compared to the control.

Treatments (applied at panicle emergence)	Biotype				Mean
	Biloela 1	Biloela 2	Toobeah	Jandowae	
Days to plant maturity (days earlier than control)					
Heat and water stress	87 (-20)	91 (-18)	96 (-19)	99 (-19)	**93 d (-19)**
Heat stress	90 (-18)	92 (-17)	99 (-16)	101 (-17)	**95 c (-17)**
Water stress	99 (-8)	101 (-8)	104 (-11)	108 (-10)	**103 b (-9)**
Control	107	109	115	118	**112 a**
Mean	**96 d**	**98 c**	**104 b**	**106 a**	
Filled primary seeds/plant (% reduction compared to control)					
Heat and water stress	142 d (-25)	119 d (-37)	155 d (-30)	120 c (-36)	**134 d (-32)**
Heat stress	147 c (-22)	125 c (-34)	167 c (-24)	123 c (-34)	**140 c (-29)**
Water stress	157 b (-17)	164 b (-13)	196 b (-11)	174 b (-7)	**173 b (-12)**
Control	189 a	189 a	220 a	187 a	**196 a**
Mean	**159 b**	**149 c**	**184 a**	**151 c**	
Filled secondary seeds/plant (% reduction compared to control)					
Heat and water stress	131 d (-28)	114 d (-37)	153 d (-28)	110 d (-39)	**127 d (-33)**
Heat stress	139 c (-23)	116 c (-36)	158 c (-25)	116 c (-35)	**132 c (-30)**
Water stress	150 b (-17)	157 b (-14)	188 b (-11)	167 b (-7)	**165 b (-13)**
Control	181 a	182 a	212 a	179 a	**189 a**
Mean	**150 b**	**142 c**	**178 a**	**143 c**	
1000 primary seed weight (g) (% reduction compared to control)					
Heat and water stress	18.79 c (-53)	25.65 d (-39)	29.98 d (-35)	33.44 c (-32)	**26.97 d (-39)**
Heat stress	19.84 c (-51)	28.83 c (-32)	32.68 c (-30)	33.90 c (-31)	**28.81 c (-35)**
Water stress	34.08 b (-15)	32.47 b (-23)	38.51 b (-17)	43.34 b (-12)	**37.10 b (-17)**
Control	40.19 a	42.28 a	46.36 a	49.03 a	**44.47 a**
Mean	**28.23 d**	**32.31 c**	**36.88 b**	**39.93 a**	
1000 secondary seed weight (g) (% reduction compared to control)					
Heat and water stress	10.57 c (-53)	14.43 d (-39)	16.87 d (-37)	18.81 c (-33)	**15.17 d (-40)**
Heat stress	11.16 c (-50)	16.22 c (-33)	18.38 c (-31)	19.07 c (-32)	**16.21 c (-36)**
Water stress	19.17 b (-16)	18.26 b (-24)	21.66 b (-19)	24.38 b (-13)	**20.87 b (-18)**
Control	22.61 a	23.78 a	26.08 a	27.58 a	**25.01 a**
Mean	**15.88 d**	**18.17 c**	**20.75 b**	**22.46 a**	

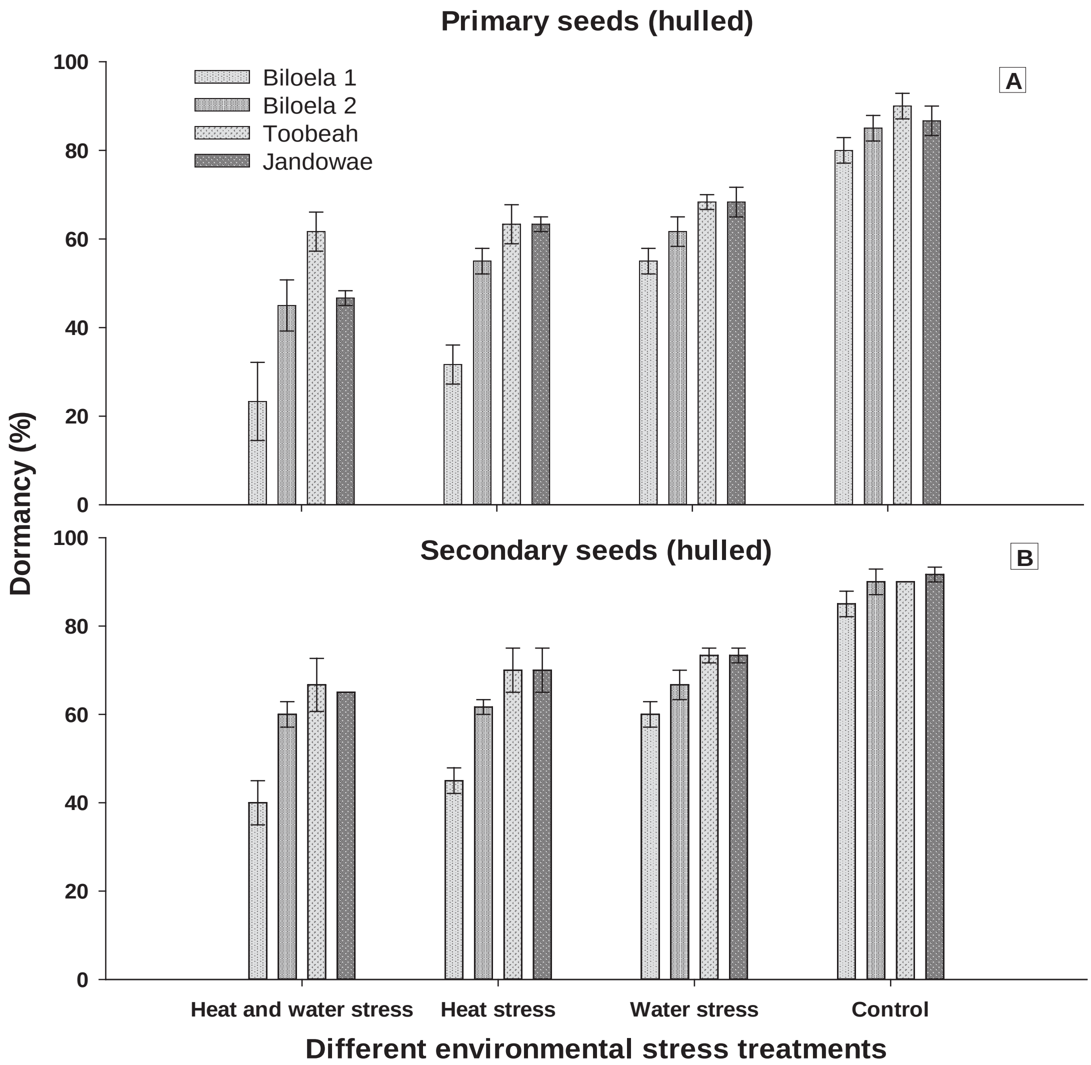


Figure 1. Effect of heat and soil water stress treatments applied at panicle emergence on percentage seed dormancy of freshly harvested (A) primary and (B) secondary hulled seeds of four *Avena sterilis* ssp. *ludoviciana* biotypes incubated at constant 9°C under 12/12 hour light/dark condition for 42 days. Error bars represent ± standard errors of the mean of three replicates.

Conclusion

The various heat and water stress treatments imposed in our study showed that wild oats plants grown under these environmental conditions are likely to mature earlier and produce fewer, smaller sized seeds that are less dormant. Given climate change forecasts for the NGR are for hotter and drier growing season, this is likely to impact on the growth, seed production and seed characteristics of wild oat populations in this region. These changes in climatic conditions will, therefore, affect the population dynamics and management strategies of wild oats. For example, hot and dry conditions will shorten the maturity time of wild oats and produce seeds with less dormancy, meaning they can potentially germinate rapidly under suitable conditions at the time of next winter crop planting in no/minimum tillage-based conservation cropping systems. Additionally, early maturity can lead to early shedding of maximum wild oats seeds to the ground before crop harvest, which is a concern in terms of harvest weed seed control. Occasional non-chemical control interventions such as the use of strategic tillage may be useful by burying shattered seeds at greater depth from where they cannot emerge. This may be a useful control tactic given the data indicates that such seeds are less well adapted for long-term survival in the soil seedbank. To assess the impact of the harvest weed seed control technique for controlling wild oats, future research should be conducted on comparing the relative maturity between wild oats and crop types and crop cultivars. To improve control of wild oats and to reduce their impact on crop production, weed management tactics and strategies will need to adapt to the changing environment and changing ecology of these weeds.

Acknowledgements

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.

References

Adkins SW, Loewen M, Symons SJ (1987). Variation within pure lines of wild oats *(Avena fatua)* in relation to temperature of development. Weed Sci 35:169-172.

CSIRO (Centre for Scientific and Industrial Research Organization) (2011). Climate change: science and solutions for Australia. Cleugh H, Smith MS, Battaglia M, Graham P (eds). CSIRO publishing, Collingwood, Victoria.

Llewellyn RS, Ronning D, Ouzman J, Walker S, Mayfield A, Clarke M (2016). Impact of Weeds on Australian Grain Production: the cost of weeds to Australian grain growers and the adoption of weed management and tillage practices. Report for GRDC. CSIRO, Australia.

Nugent T, Storrie A, Medd R (1999). Managing wild oats. The Handbook Jointly held by CRC for Weed Management Systems and Grains Research and Development Corporation' ISBN 1875477438, October 1999.

Quail PH and Carter OG (1968). Survival and seasonal germination of seeds *of Avena fatua* and *A. ludoviciana*. Aust J Agríc Res 19:721-729.

Contact details

Mohammad Ali
The University of Queensland
School of Agriculture and Food Sciences
Gatton QLD 4343
Ph: 07 5460 1321
Email: mohammad.ali@uq.edu.au

Glyphosate and new cases of resistance- Narrabri

Peter Boutsalis[1,2], Michael Walsh[3], Christopher Preston[1] and John Broster[4]

[1]School of Agriculture, Food & Wine, University of Adelaide
[2]Plant Science Consulting P/L
[3] School of Life and Environmental Sciences, University of Sydney
[4] Graham Centre for Agricultural Innovation (Charles Sturt University and NSW Department of Primary Industries), Charles Sturt University

Keywords

optimising glyphosate performance, annual ryegrass, sowthistle, clethodim, new cases of resistance, NSW random weed survey, germination patterns

GRDC codes

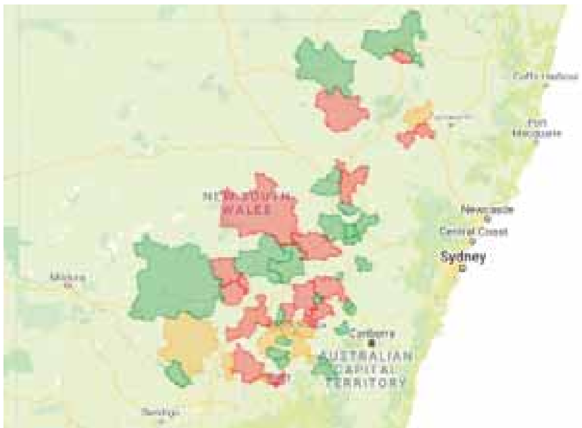
UCS00020, US00084

Take home messages

- Increased glyphosate resistant annual ryegrass detected in NSW
- Glyphosate performance can be optimised through attention to application variables such as temperature, plant stress and formulation
- New cases of herbicide resistance have been detected

Incidence of glyphosate resistance in NSW

Bayer CropScience provide access to a significant database (Resistance tracker) combining data from national random weed surveys and commercial testing companies (https://www.crop.bayer.com.au/tools/mix-it-up/resistance-tracker). This tracker tool enables the searching of resistance to numerous weed species by postcode and year, with data presented from 2003 (Figure 1).

2003	2009
	

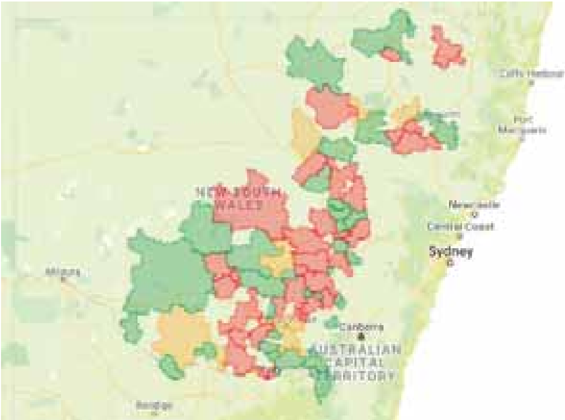
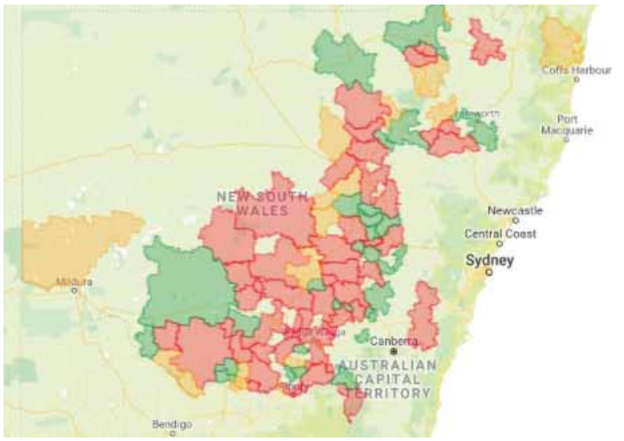
2015	2020
	

Figure 1. Occurrence of glyphosate resistance in annual ryegrass in NSW in 2003, 2009, 2015 and 2020. **Dark green shading** = postcode regions where testing has not detected glyphosate resistance in ryegrass, **orange shading** = postcodes where glyphosate resistance is developing and **red shading** = postcodes where resistance has been detected

2020 season: The early break in 2020 across most southern cropping regions resulted in an opportunity for knockdown weed control. Multiple applications of glyphosate and paraquat were targeted at multiple flushes of weeds, in particular ryegrass, from early autumn to prior to sowing. Plants surviving glyphosate from WA, SA, Vic and NSW were sent to Plant Science Consulting for testing using the Quick-Test method to verify whether herbicide resistance had contributed to survival in the field. The data presented in Figure 2 indicates that 43%, 70% and 79% of ryegrass samples sent from SA, Vic and NSW in 2020 respectively, were confirmed resistant to glyphosate. This highlights that in a majority of cases in NSW, glyphosate resistance has contributed to reduced control in the paddock.

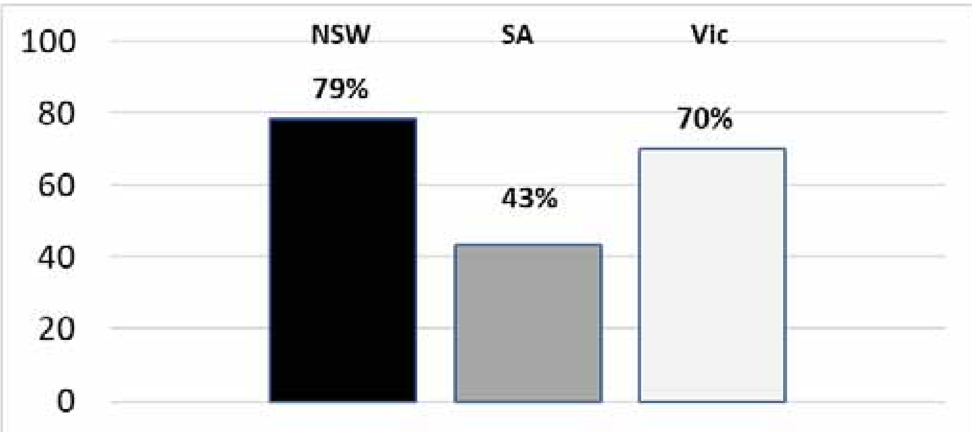


Figure 2. Percent (%) resistance to glyphosate confirmed in farmer ryegrass samples originating from 83 NSW, 37 SA and 74 Vic cropping paddocks treated with glyphosate in autumn 2020. Testing conducted by Plant Science Consulting using the Quick-Test

Discrepancy between resistance testing and paddock failures to glyphosate

In some cases, plants that survived glyphosate in the paddock are not resistant. Reasons for the discrepancy between the paddock and a resistance test can include poor application or application onto stressed plants, incorrect timing, sampling plants that were not exposed to glyphosate, antagonistic tank mixes, inferior glyphosate formulation, poor water quality, incorrect adjuvants, or a combination of the above.

Evolution of glyphosate resistance

Glyphosate was first registered in the 1970's and rapidly became the benchmark herbicide for non-selective weed control. Resistance was not detected until 1996 in annual ryegrass in an orchard in southern NSW (Powles et. al. 1998). Only a few cases of resistance were detected in the following decade (refer to Bayer Resistance Tracker app). The fact that it required decades of repeated use before resistance was confirmed indicated that the natural frequency of glyphosate resistance was initially very low. At the current time there are over a dozen species that have developed resistance to glyphosate in Australia (https://www.croplife.org.au/resources/programs/resistance-management/herbicide-resistant-weeds-list-draft-3/). The most important species in NSW are ryegrass, sowthistle, barnyard grass, fleabane and feathertop Rhodes grass. Ryegrass and sowthistle will be discussed below.

Sowthistle

Differences in the level of control between glyphosate formulations in a glyphosate-resistant sowthistle population from NSW was identified (Figure 3). This finding highlights that significant differences in activity between glyphosate formulations may occur on both susceptible and glyphosate resistant individuals.

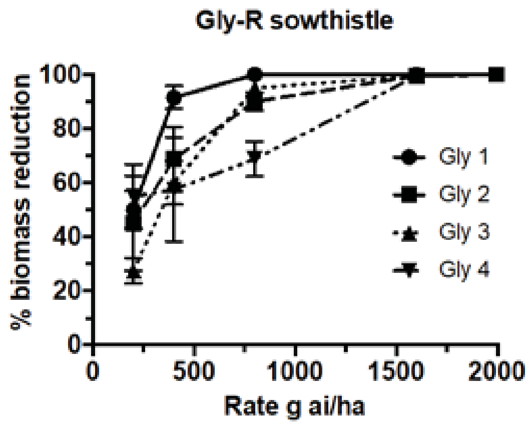


Figure 3. Efficacy of four glyphosate products on control of glyphosate resistant sowthistle as confirmed by outdoor pot trials by Plant Science Consulting

Growth stage and glyphosate rate

Plant growth stage can play an important role in weed control. Even in resistant populations, improved control can often be achieved when targeting younger growth stages. Younger plants tend to have thinner cuticles than older plants improving the speed of uptake. The effect of growth stage and glyphosate rate was investigated in a field trial in NSW on one susceptible and two glyphosate resistant sowthistle populations by Tony Cook, DPI Tamworth (Table 1). Increased control of glyphosate resistant sowthistle was observed at younger growth stages and at higher rates.

Table 1. First cases of confirmed glyphosate resistant sowthistle from Liverpool plains. Data presented as percent biomass reduction at three growth stages. Fallow spray timings from early to late summer. Data courtesy of Tony Cooke, DPI, Tamworth.

Glyphosate rate (g ai/ha)	Growth stage: Early rosette 10cm	Growth stage: Early bolting	Growth stage: Mid-flowering
Susceptible sowthistle- (% biomass reduction)			
360	79	76	0
720	100	81	33
Resistant sowthistle biotype "Yellow" - (% biomass reduction)			
360	55	27	0
720	97	0	0
Resistant sowthistle biotype "CRK" - (% biomass reduction)			
360	64	7	0
720	80	35	5

Application conditions

The effect of temperature on control of glyphosate resistant sowthistle from NSW was recently investigated. Initial trials have confirmed greater control with glyphosate at lower temperatures, particularly of resistant biotypes (Table 2). These findings suggest that applying glyphosate at lower temperatures can improve control of glyphosate resistant sowthistle. At lower temperatures glyphosate remains in liquid form on plant surfaces longer leading to greater uptake, particularly under higher humidity. This allows more hours on the leaf for glyphosate to penetrate. Maximising glyphosate uptake is therefore likely to improve weed control and factors such as lower temperature and higher humidity influence uptake. A pot trial investigating the effect of 20°C and 30°C on glyphosate efficacy identified a higher LD_{50} required at 30°C . This indicates that approximately 2.5x more glyphosate was required to control the resistant populations at 30°C, as opposed to the same populations grown at 20°C

Table 2. Effect of temperature in control of four biotypes of sowthistle from NSW with Glyphosate 540g/L. Data is LD_{50}= dose required to kill 50% of the population.

Biotypes	Resistance level	LD_{50} (g a.i/ha)	
		20°C	30°C
Yellow	strong	439	962
Crocket	strong	389	919
White	weak	132	389
GI	susceptible	135	152

Factors affecting glyphosate performance

There are several contributing factors for the increasing frequency of glyphosate resistance with generally more than one factor responsible. Lower application rates can increase the selection for resistance, particularly in an obligate outcrossing species such as ryegrass, resulting in the accumulation of weak resistance mechanisms to create individuals capable of surviving higher rates. This has been confirmed by Dr Chris Preston where ryegrass hybrids possessing multiple resistance mechanisms were generated by crossing parent plants with different resistance mechanisms.

Other factors that can select for glyphosate resistance by reducing efficacy include:

1. Using low quality glyphosate products and surfactants
2. Mixing glyphosate with too many other active ingredients resulting in antagonism, particularly in low water volumes
3. Using low quality water, particularly hard water. Glyphosate is a weak acid and binds to positive cations (i.e. magnesium, calcium and bicarbonate) that are in high concentration in hard water (i.e. >200 ppm)
4. Applying glyphosate during periods of high temperature and low humidity, resulting in the rapid loss of glyphosate in solution from leaf surfaces and thereby reducing absorption
5. Translocation of glyphosate in stressed plants can be reduced. Maximum glyphosate efficacy relies on translocation to the root and shoot tips. While this occurs readily in small seedlings, in larger plants, glyphosate is required to translocate further to the root and shoot tips to provide high levels of control
6. Shading effects reducing leaf coverage resulting in sub-lethal effects
7. As glyphosate strongly binds to soil particles, application onto dust covered leaves can reduce efficacy
8. Application factors such as speed and nozzle selection, boom height can reduce the amount of glyphosate coverage
9. A combination of the above factors can reduce control thereby increasing the selection for resistance.

Optimising glyphosate performance

The selection of glyphosate resistance can be reduced by considering the points above. A number of important pathways to improve glyphosate performance include:

Avoid applying glyphosate under hot conditions

A trial spraying ryegrass during the end of a hot period and a following cool change was conducted in October 2019. Ryegrass growing in pots was sprayed at 8am, 1pm and 8pm with temperature and Delta T recorded prior to each application. Control of well hydrated plants ranged between 0% and 40% when glyphosate was applied during hot weather (30 to 32.5°C) and high Delta T (14 to 16.7), with the lowest control when glyphosate was applied at midday (Figure 4). In contrast, glyphosate applied under cool conditions just after a hot spell resulted in significantly greater control (65%-80%), indicating that plants can rapidly recover from temperature stress provided moisture is not limiting, eg. after rainfall.

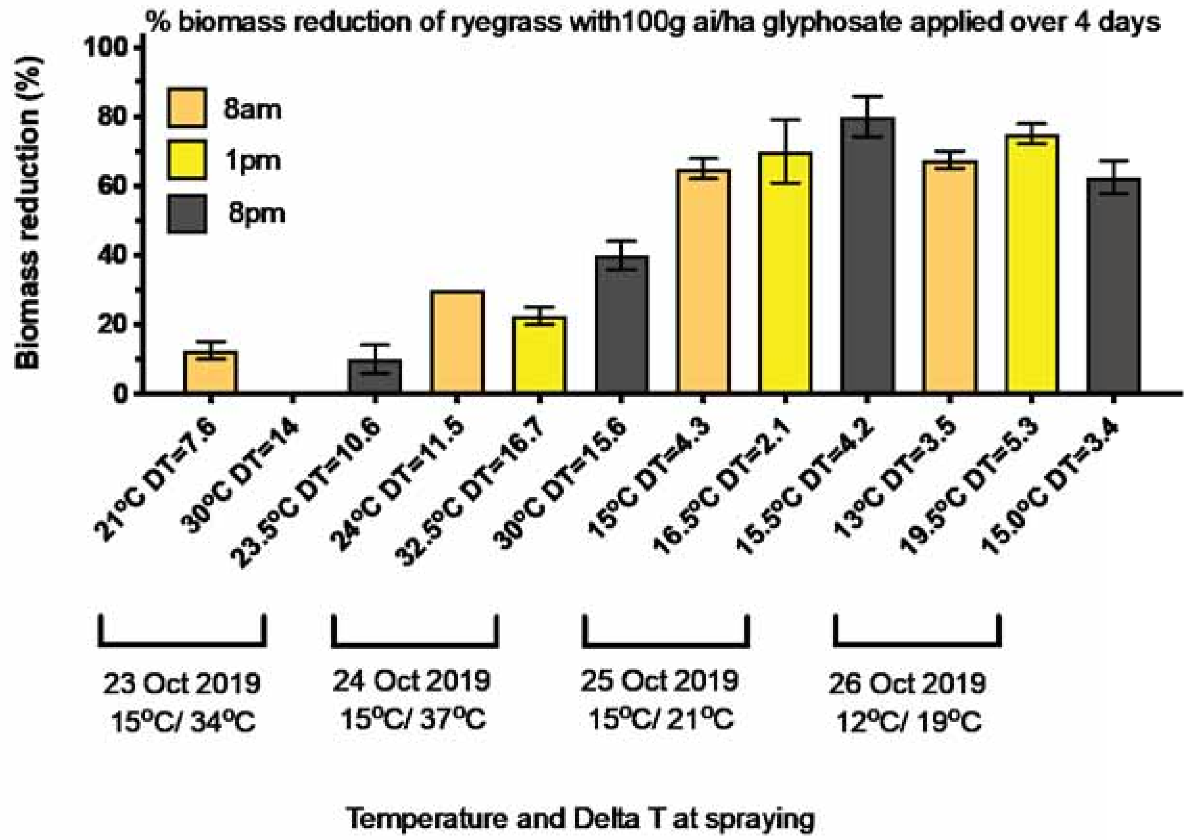


Figure 4: Effect of temperature & Delta T on glyphosate for ryegrass control
(A sub lethal rate was used to differentiate between treatment differences. Plants were grown and sprayed under optimum conditions).

Improving water quality and glyphosate activity by using ammonium sulfate (AMS)

The addition of AMS has several functions. One is to soften water by combining to positively charged ions such as magnesium and calcium common in hard water. The negative charged sulfate ions combine with the positive cations preventing them from interacting with glyphosate and reducing its solubility and leaf penetration. Additionally, AMS has been shown to independently improve glyphosate performance, as the ammonium ions can work with glyphosate to increase cell uptake. In a pot trial conducted with soft water, ammonium sulfate was shown to significantly improve control of ryegrass with 222ml/ha (100g ai/ha) of glyphosate 450 (Figure 5). As a general rule, growers using rainwater (soft) should consider 1% liquid AMS, if using hardwater (i.e. bore, dam) 2% AMS. The addition of a wetter resulted in a further improvement in control.

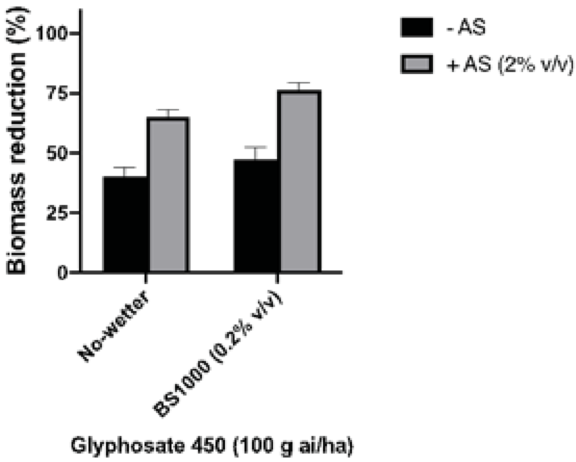


Figure 5: Effect of ammonium sulfate (AS) and wetter on glyphosate for ryegrass control
(A sub lethal rate was used to differentiate between treatment differences. Plants were grown and sprayed under optimum conditions).

Herbicide activity can vary at different growth stages

In a pot trial investigating the effect of glyphosate at 4 ryegrass growth stages (1-leaf to 4-tiller), good control was achieved at the 3 older growth stages but not on 1-leaf ryegrass (Figure 6). Most glyphosate labels do not recommend application of glyphosate on 1-leaf ryegrass seedlings because they are still relying on seed reserves for growth. As a consequence, very little glyphosate moves towards the roots.

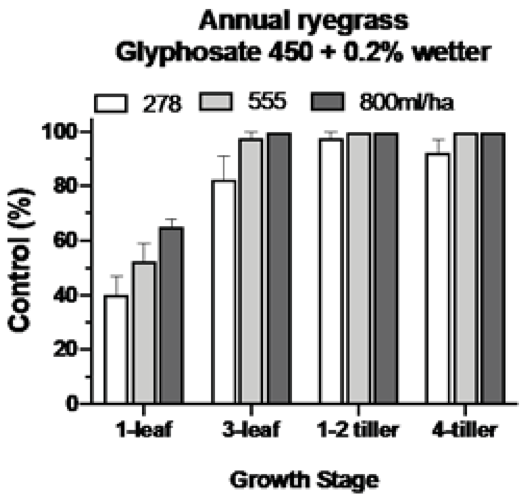


Figure 6: Effect of ryegrass growth stage on glyphosate activity
(A sub lethal rate was used to differentiate between treatment differences. Plants were grown and sprayed under optimum conditions).

Double knock

A double knock strategy is defined as the sequential application of two weed control tactics directed at the same weed cohort (germination). The most common double knock strategy is glyphosate followed by paraquat. This has been widely adopted to prevent or combat glyphosate resistance in several weed species, including ryegrass. The first 'knock' with glyphosate controls the majority of the population, with the second 'knock' (paraquat) intended to kill any individuals that have survived glyphosate. Trial work conducted by Dr Christopher Preston (Figure 9) showed that control was optimised when the paraquat was applied 1-5 days after the glyphosate for two glyphosate resistant ryegrass populations. However optimal timing depends on weed size and growing conditions, with at least 3-5 days often being required for full glyphosate uptake and translocation, especially in larger plants. In this study, when the glyphosate resistant plants were left for 7 days before the paraquat application they can stress, resulting in the absorption of less paraquat, reducing control with the second tactic. If growing conditions are poor or plants large, the stress imposed by glyphosate maybe further delayed.

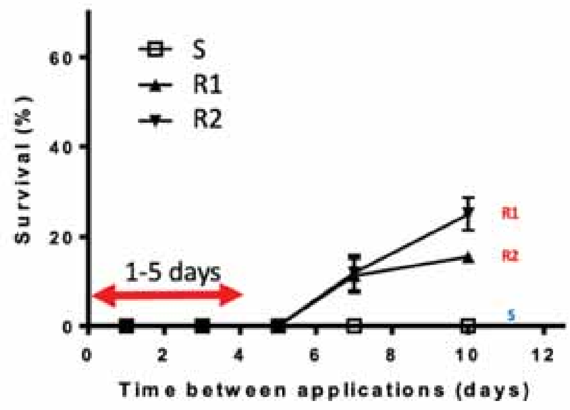


Figure 7: Double knock timing. Glyphosate applied onto a susceptible (S) and two glyphosate resistant ryegrass biotypes (R1 & R2) followed by paraquat 1, 3, 5, 7 and 10 DAA. Trial work conducted by Dr Christopher Preston (The University of Adelaide).

New resistance data

Phalaris

In the 2020 season, 13 phalaris plant samples from populations in paddocks collected from NSW and Qld were tested. Resistance to Group A and/ or B herbicides was confirmed in 10 samples (Table 3).

Table 3. Percent (%) survival of phalaris from NSW and Qld to herbicides. Testing of plants growing in paddocks in 2020 using the Quick test was conducted by Plant Science Consulting. (* herbicide not tested)

Town	**State**	Pinoxaden	Clethodim	Clodinafop	Haloxyfop	imazamox/ imazapyr	mesosulfuron
Widgelli	NSW	*	0	*	*	*	*
Yenda	NSW	*	85	*	*	*	*
Moree	NSW	25	10	40	40	*	0
Moree	NSW	*	*	*	*	*	0
Moree	NSW	65	45	*	50	*	0
Moree	NSW	20	*	*	*	40	50
Goondiwindi	QLD	100	80	100	100	0	0
Bellata	NSW	*	*	*	*	30	60
Croppa Creek	NSW	*	50	*	90	0	*
Pallamallawa	NSW	90	*	*	*	*	*
North Star	NSW	0	0	20	20	60	45
Tulloona	NSW	75	75	*	75	0	15
Tulloona	NSW	0	0	*	0	0	0

Paterson's curse

In the 2019 season, seeds from Paterson's curse that had not been controlled with glyphosate in cotton paddock near Hillston NSW was tested for glyphosate resistance. Survival to glyphosate was confirmed in two trials at 45% survival to 690 g ai/ha at the label rate which controlled a susceptible control population. This is the first reported case of glyphosate resistance in Paterson's curse.

Echinochloa crus-galli

In a 2019 rice crop near Murrambi NSW, poor control of *Echinochloa crus-galli* with Barnstorm® was detected. Seed was sent to Plant Science Consulting for resistance testing in 2020. Strong resistance to Barnstorm (FOP) and lower resistance to Aura® (DIM) was detected, confirming that resistance had contributed to reduced control of *Echinochloa* in the rice field (Figure 8).

This is the first case of Group A resistance in *Echinochloa crus-galli* detected in Australia.

Wild oats

Over 100 samples of wild oats were received in 2020, some as plants during the cropping season and most as seed tests currently being testing. The most common resistance detected is to Group A herbicides. No resistance to triallate has been confirmed. Flamprop-methyl, previously registered as Mataven® and now available as several generic brands, is being included by some growers in their resistant test options.

Ryegrass

The only new case of resistance in annual ryegrass is to paraquat. About half a dozen populations from SA, Vic and WA have been confirmed. Most cases are from situations where paraquat is frequently applied such as lucerne and white clover seed production, fencelines and non-cropped areas. The two Victorian cases however are from cropping paddocks. No cases (to my knowledge) have been detected in NSW. The fact that paraquat resistance has been confirmed in annual ryegrass is of great concern.

Incidence of herbicide resistance in northern region winter annual weeds

Annual ryegrass is the dominant weed of the northern cropping region, despite little or no occurrence of this species in Queensland. Across NSW approximately 70% of annual ryegrass populations are resistant to one or more herbicides (Figure 8), therefore as this species continues to migrate northwards into Queensland it will be resistant populations that establish in these cropping regions.

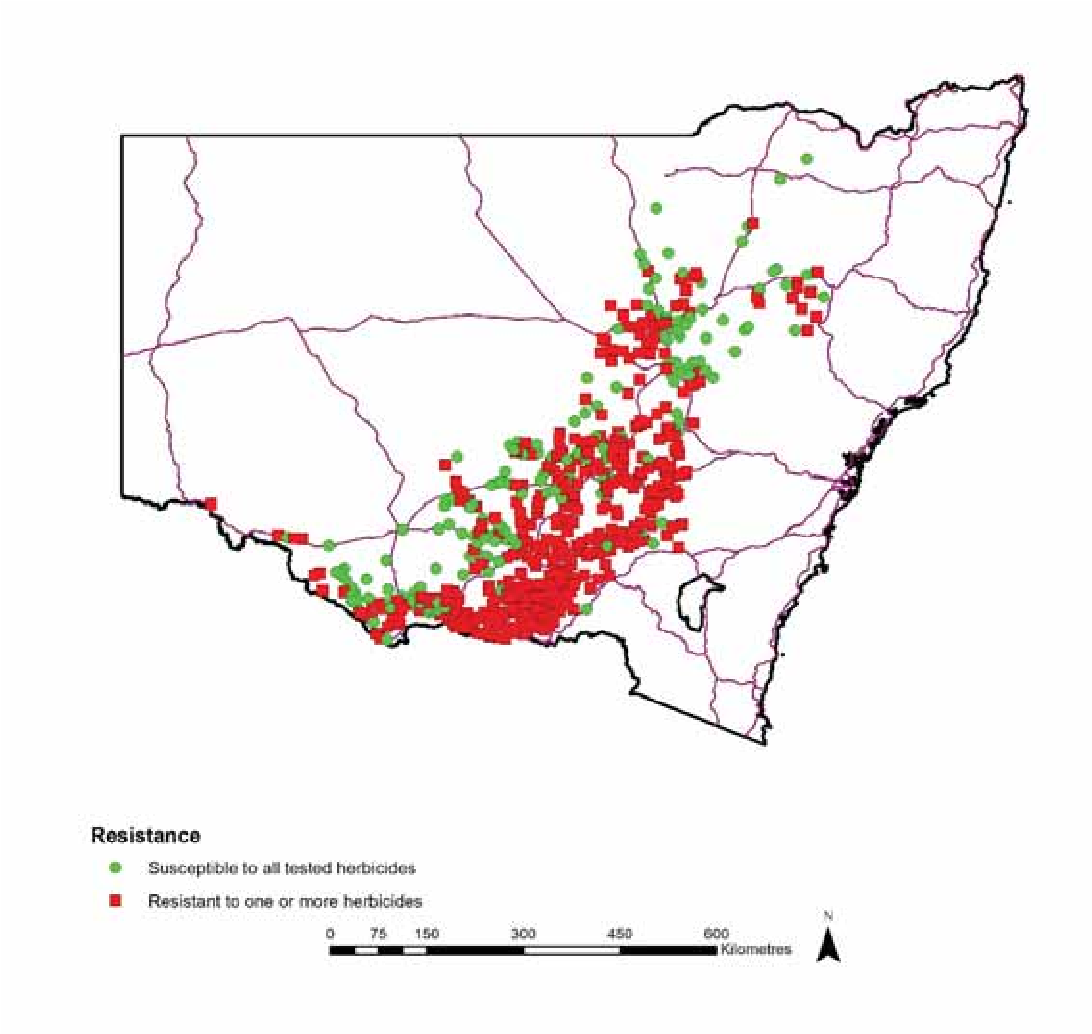


Figure 8. Distribution of northern region annual ryegrass populations that are susceptible (green circles) or resistant (red squares) to one or more herbicides.

The majority of annual ryegrass populations in NSW are resistant to Group A 'fop' and Group B herbicides with some variability between the surveyed regions (Table 4). No populations have been found that are resistant to the newer pre-emergent herbicides, although this has been reported in other states. Of particular concern is the number of populations resistant to glyphosate in some regions.

Table 4. Frequency of herbicide resistance in annual ryegrass populations collected in NSW random surveys (2015 to 2019) (Resistance >20% survival)

	NSW (2015 - 2019)	2015 western NSW	2016 NSW northern	2016 NSW plains	2017 southern NSW	2018 NSW slopes	2019 eastern NSW
	Populations resistant (%)						
diclofop	59	16	32	65	84	77	92
clethodim	2	1	1	1	3	0	12
sulfometuron	50	30	22	35	74	70	82
imazamox/imazapyr	47	8	22	39	75	76	83
trifluralin	1	2	0	0	1	1	2
prosulfocarb + S-metolachlor	0	0	0	0	0	0	0
pyroxasulfone	0	0	0	0	0	0	0
glyphosate	5	6	5	0	7	3	14
Populations screened	608	117	94	111	128	105	53

Wild oat is the next most resistant weed of the northern cropping region with approximately 40% of populations resistant to one or more herbicides (Figure 9). The majority of this resistance is to the Group A herbicide clodinafop (Table 5). Resistance to this herbicide was similar in northern NSW (34%) and Queensland (33%). The highest frequencies of resistance were found in the eastern NSW area where 46% were resistant to clodinafop, with the extent of resistance in Queensland populations higher than all NSW regions except for the eastern area (Table 5).

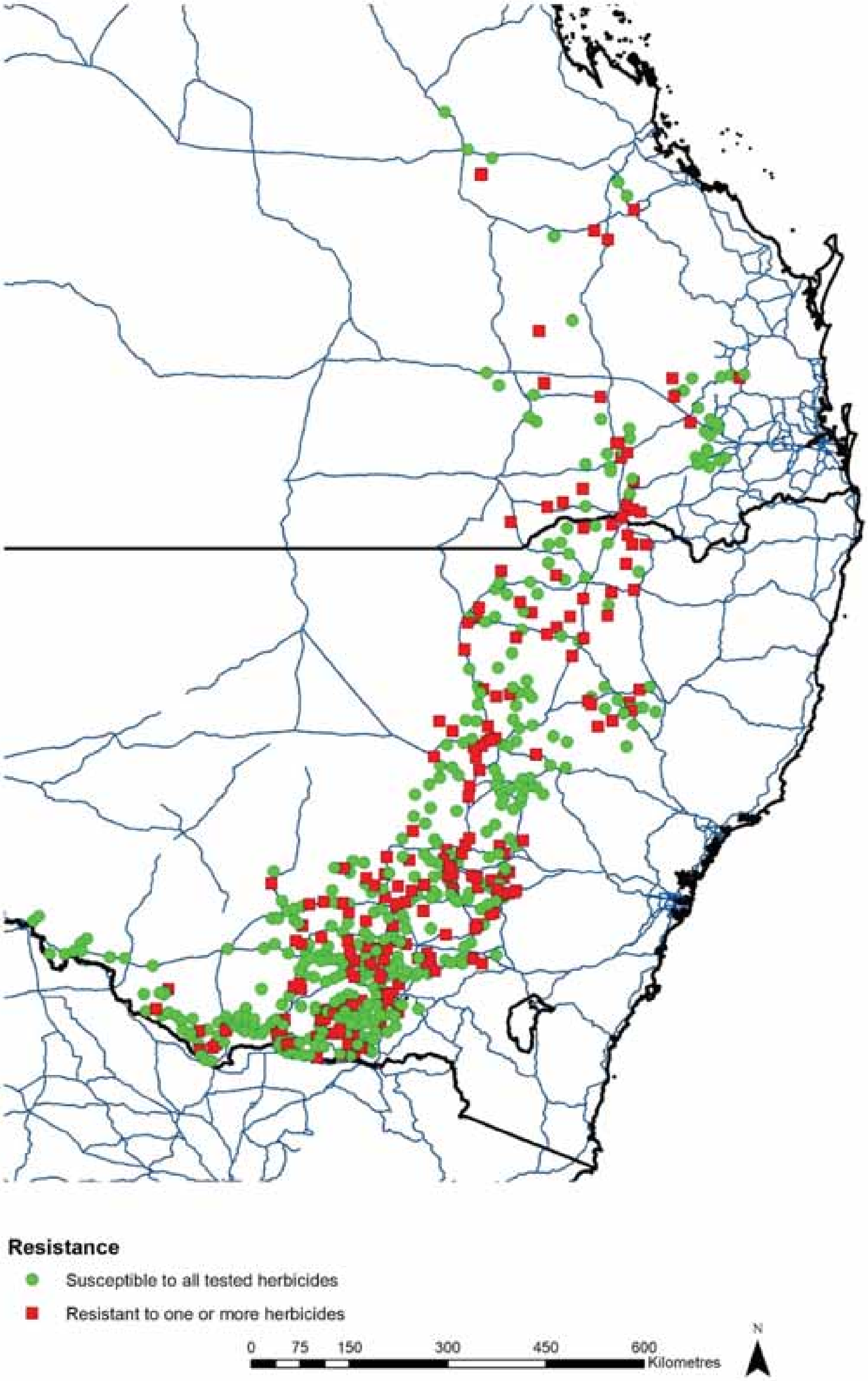


Figure 9. Distribution of northern region wild oat populations that are herbicide susceptible (green circles) or resistant (red squares).

Table 5. Frequency of herbicide resistant wild oat populations randomly collected in recent surveys of the northern cropping region. (Resistant >20% survival) (* = herbicide not screened)

Herbicide	NSW (2015 - 2019)	Qld (2016)	2015 western NSW	2016 NSW northern	2016 NSW plains	2017 southern NSW	2018 NSW slopes	2019 eastern NSW
	Herbicide resistant populations (%)							
Clodinafop	29	33	12	34	31	30	30	46
Clethodim	1	0	0	0	3	0	0	4
Iodosulfuron	4	3	6	4	3	0	0	18
Triallate	0	0	0	0	0	0	0	0
Glyphosate	0	0	*	0	*	0	0	0
Populations screened	511	72	94	122	70	104	93	28

Among the other species collected during the random survey resistance was much lower with no populations of barley grass or brome grass resistant to Group A herbicides and very rare resistance to Group B herbicides. Group F and I resistance was common for wild radish (44% and 29% respectively) and Group B SU resistance in Indian hedge mustard (25%) (Table 6). Of concern is that 3% of barley grass populations were resistant to paraquat (Table 6).

Table 6. Extent of herbicide resistance in populations of the other species collected in random surveys of the northern cropping region (NSW and Qld) (Resistance >20% survival, * herbicide not tested or not applicable for species)

Herbicide group	Barley grass	Brome grass	Wild radish	Indian hedge mustard
	Herbicide resistant populations (%)			
Quizalofop	0	0	*	*
Clethodim	0	0	*	*
Mesosulfuron	1	7	*	*
Chlorsulfuron	*	*	3	25
Imazamox/ Imazapyr	*	2	3	4
Atrazine	*	*	3	0
Diflufenican	*	*	44	6
2,4-D Amine	*	*	29	2
Gramoxone	3	*	*	*
Glyphosate	*	0	0	0
Samples	133	110	33	59

Incidence of herbicide resistance in northern region summer annual weeds

An additional survey collected weed species across northern NSW and Queensland in summer 2016/17. The species collected during this survey included awnless barnyard grass, feathertop Rhodes grass, fleabane and some additional sow thistle samples. These samples were screened for resistance to glyphosate with a significant percentage of populations for all species except for sow thistle resistant to this herbicide (Table 7).

Table 7: Extent of glyphosate resistance for weed species collected in 2016 summer survey (Includes sow thistle collected in northern NSW and Queensland winter survey)

	Northern NSW		Queensland	
	% Resistant	Populations tested	% Resistant	Populations tested
Awnless barnyard grass	0	5	37	37
Feathertop Rhodes grass	50	2	70	60
Fleabane	100	25	100	36
Sow thistle	7	45	3	62

Summary

Ryegrass blowouts in 2020 were a result of large seedbanks, reduced weed control tactics and herbicide resistance. Using multiple tactics, including new mode of action pre-emergent herbicides, can improve ryegrass control.

While paraquat resistance remains very low, glyphosate resistance in ryegrass is increasing at a rapid rate. The early break in autumn 2020 resulted in the testing of about 200 ryegrass populations prior to sowing with over half confirmed resistant to glyphosate. Decades of strong selection pressure resulting from repeated use coupled with application under suboptimum conditions has played a major role. More efficient use of glyphosate combined with effective IWM strategies is required to reduce further increases in resistance in ryegrass and sowthistle.

Acknowledgements

The information for the random weed surveys was undertaken as part of GRDC projects UCS00020 and US00084

Contact details

Peter Boutsalis, Plant Science Consulting P/L
Herbicide resistance website: www.plantscienceconsulting.com.au
@PBoutsalis
Peter Boutsalis, University of Adelaide, Waite Campus, Glen Osmond SA 5064
Email: peter.boutsalis@adelaide.edu.au

John Broster
Charles Sturt University, Wagga Wagga, NSW
Email: JBroster@csu.edu.au

Chickpea yield gaps – how much yield potential could we be losing in a dry season, and why?

Allan Peake[1], Zvi Hochman[2], Heidi Horan[2], Elizabeth Meier[2], Brook Anderson[1] and Javier Navarro Garcia[2]

[1] CSIRO Agriculture and Food, Queensland Bioscience Precinct, 306 Carmody Road, St Lucia, Qld, 4067
[2] CSIRO Agriculture and Food, P.O. Box 102 (203 Tor St), Toowoomba, Qld, 4350

Key words

yield gap, pulse crops, chickpea, evaluation, management, risk, APSIM, agronomy

GRDC code

CSP1904-005RXT: 'The adaptation of pulse (chickpea and lentil) across the northern grains region.'

Take home messages

- Chickpea yield gaps tend to be larger than for cereal and canola crops
- Average chickpea yields between 1996 and 2015 for growers in the GRDC northern region (1.05 t/ha) are 1.6 t/ha below the water-limited yield potential. On average, this is costing growers $806/ha
- Average chickpea yields between 1996 and 2015 for growers in the GRDC southern region (0.82 t/ha) are 2.38 t/ha below the water-limited yield potential. On average, this is costing growers $1,200/ha
- Alternative management (narrower rows, higher plant populations and earlier sowing) was predicted to increase grain yield by 0.4 t/ha on average across monitored fields in 2019
- Preliminary recommendations are made for optimum sowing date at a range of representative locations across northern NSW.

Introduction

The yield gap is the difference between the actual yield achieved by the average grower and the water limited yield potential (Figure 1). The water-limited potential is defined as the maximum possible yield able to be grown with the optimal sowing date, current cultivars and with nutrients, pests, disease and weeds not limiting yield. Historically, yield gaps are calculated after the actual yield is observed, and potential yield is calculated using a crop growth model. When crops are grown in response to a variable climate such as that of the Australian grain zone, a period of 15 years is required to reliably capture the variability of year to year weather and hence of the extent of yield gaps in each year. Previous studies have shown that, over a wide range of seasonal conditions, well managed commercial wheat crops can reach about 80% of their potential (van Rees et al., 2014). This yield level is called the exploitable yield (Figure 1).

It is well known that Australia's grain growers are among the best in the world so how close are chickpea growers in the northern region to achieving their water-limited yield potential? How much does this vary from area to area?

An earlier GRDC-funded project (CAS00055) identified that rainfed crops in Australia achieved about half of their water-limited yield potential in 2000-2014. This was made up of 50% for wheat, 53% for canola, 40% for barley, 58% for sorghum and 39% for pulse crops (www.yieldgapaustralia.com).

As part of the GRDC funded 'Pulse Adaptation' project, we examined the latest data to see whether significant yield gaps still exist in chickpea, and if so, what might be causing them. Two types of yield gap analyses were conducted: one at a regional data scale (described below in part A), and the other at a single paddock scale (described in part B).

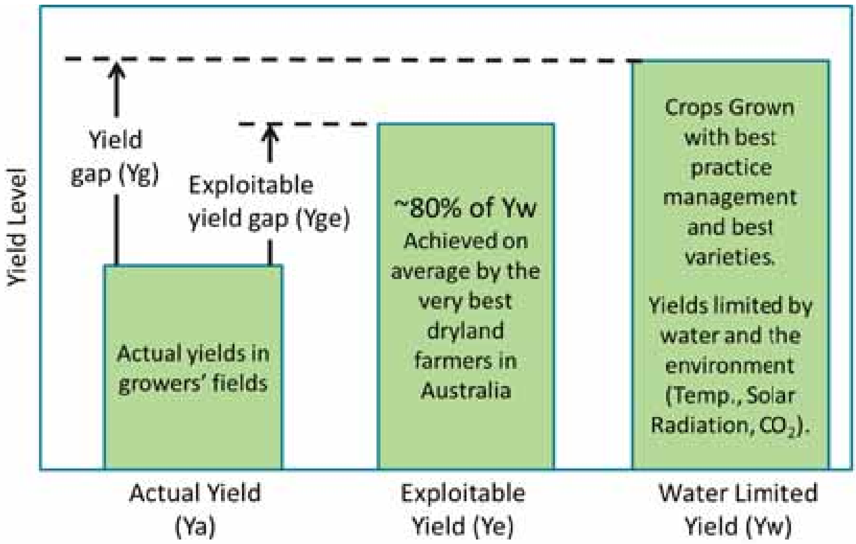


Figure 1. Actual yield, water-limited yield and exploitable yields (after Lobell et al. 2009)

A. Region scale yield gap analysis

What did we do?

For the regional analysis, we calculated the yield gap by subtracting average farm yields from the water-limited yield potential simulated with the APSIM crop growth model. Actual farm yields (Ya) were derived from the latest available ABS and ABARES sources at the statistical local area (SA2), which were averaged over the period from 1996 to 2015 to account for seasonal variability. Water-limited yield potential (Yw) is defined as the APSIM simulated yield of a crop for a given soil type using the rainfall and soil water available, but assuming that frost, diseases, pests and nutritional disorders have not affected yield.

Yw was simulated with the APSIM chickpea model for at least 5 years of the 20 year study period for the three dominant soil types within a 20 km radius of every weather station in each SA2 in the Australian cropping zone where actual yield data were available (Figure 2). All crops were simulated according to current best practice. Sowing windows varied by location: Central Queensland = 15 Apr – 7 Jun; SE Queensland and northern NSW = 15 May – 7 Jul; Victoria = 7 May – 7 Jul. For all regions a sowing opportunity required rain >= 15mm over 3 consecutive days during the sowing window. Sowing density was set at 30 plants/m^2, row spacing at 500 mm, and sowing depth at 50 mm. Two varieties were simulated: HatTrick⯑ (mid-late) and Seamer⯑ (early) for each weather station x soil combination and the highest yielding variety (average 1996-2015) was chosen to represent Yw. Sowing soil water was determined by running a 15-year wheat/chickpea continuous crop simulation prior to each year used in the yield gap simulation. We then interpolated the Yw data from each station x soil combination to their SA2s to produce the Yw map at SA2 resolution to match all the SA2s in Figure 2. This enabled us to produce a chickpea water-limited yield potential map (Figure 3).

Yield gaps (Yg, the difference between water-limited chickpea yields and actual chickpea yields) were calculated for each SA2 to produce a chickpea yield gap map (Figure 4). Finally, because areas with larger water-limited yields tend to have larger yield gaps we are also interested in the yields achieved relative to their potential, we present the relative yield (Y%) achieved by growers (Figure 5.). This is calculated as: Y% = 100 x Ya/Yw

It is important to note that the confidence we have in calculating chickpea yield gaps is lower than for crops such a wheat and sorghum. This is due to some uncertainty about the reliability of actual farm data due to gaps in the ABS and ABARES data on farm yields and in uncertainty about the accuracy of the yield predictions of the APSIM chickpea crop module.

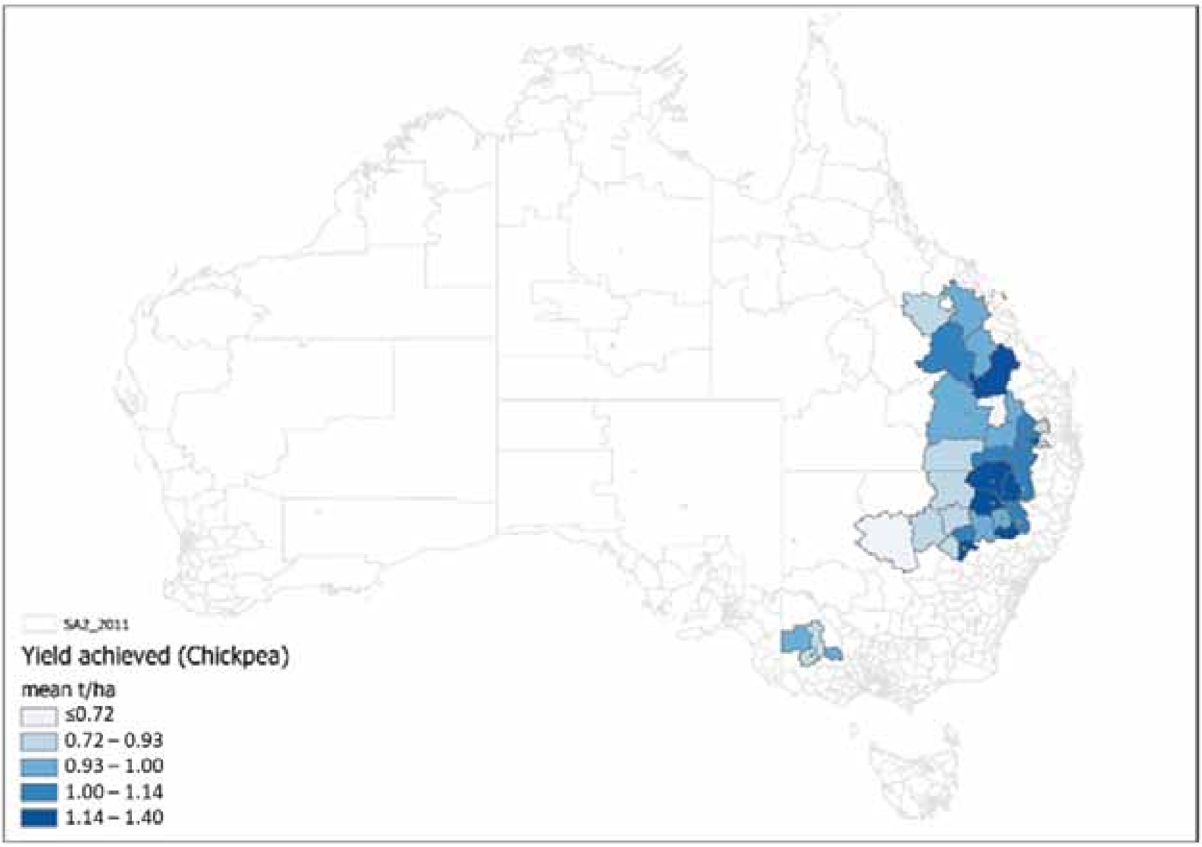


Figure 2. Actual chickpea yields (Ya) per statistical local area (SA2), averaged from 1996 to 2015.

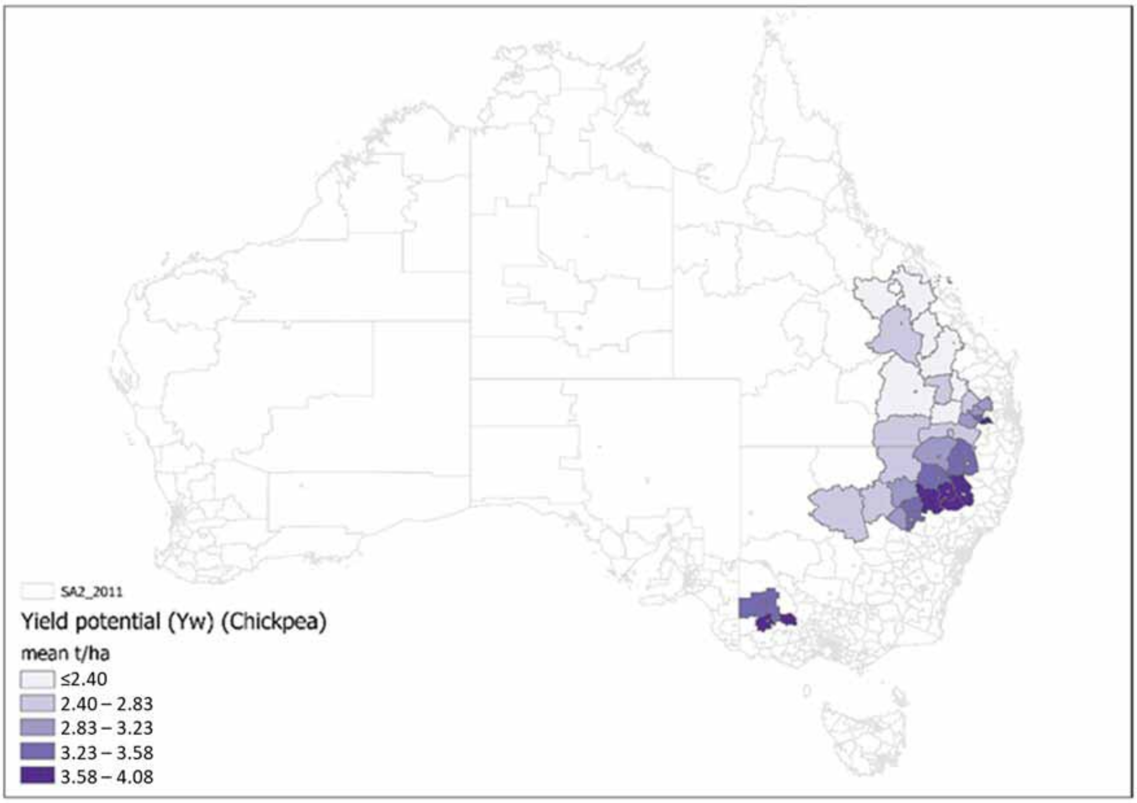


Figure 3. Simulated water-limited chickpea yields (Yw) per statistical local area (SA2), averaged from 1996 to 2015.

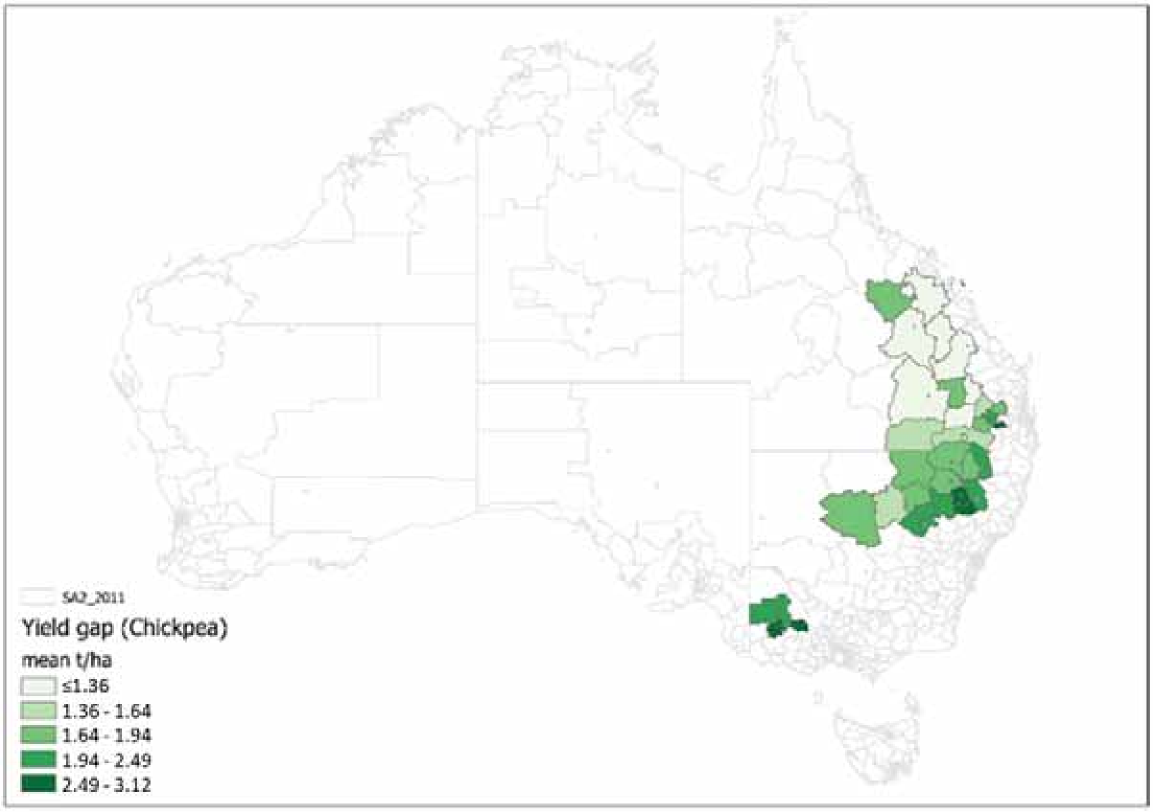


Figure 4. Chickpea yield gaps (Yg) per statistical local area (SA2), averaged from 1996 to 2015.

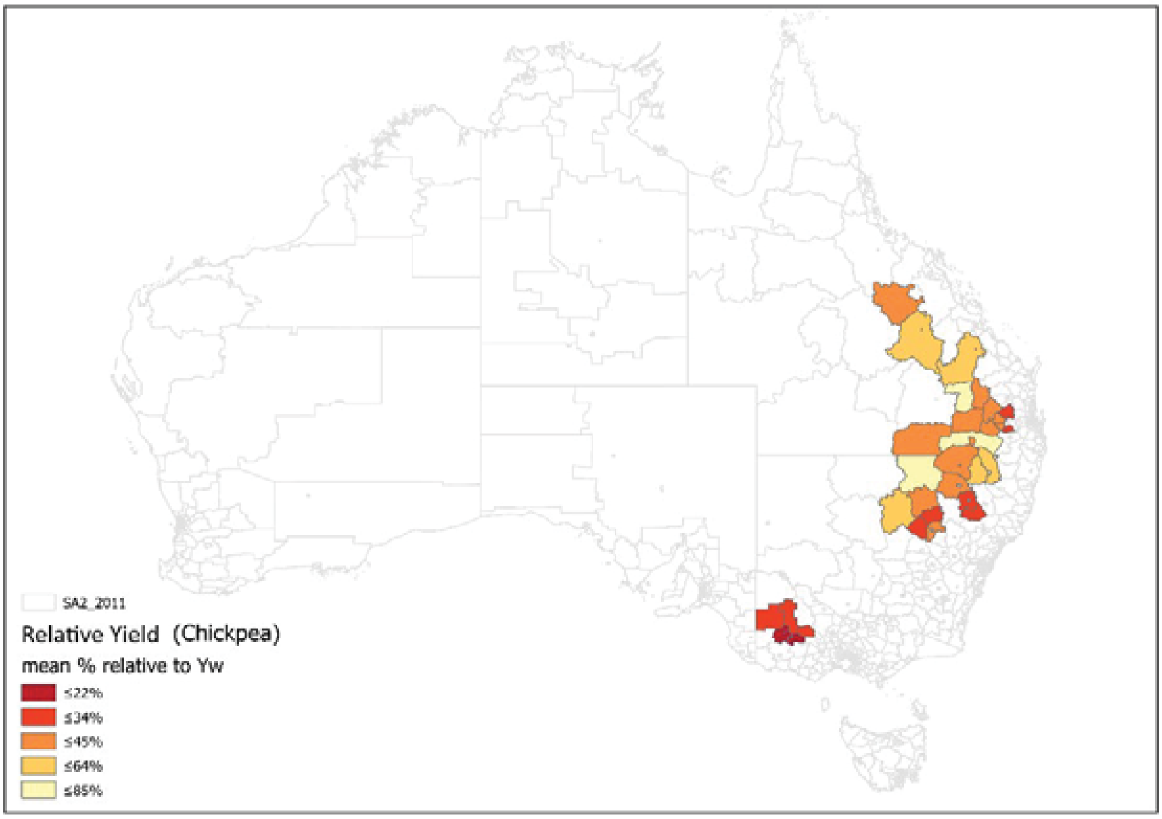


Figure 5. Chickpea relative yields (Y%) per statistical local area (SA2), averaged from 1996 to 2015.

What did we learn?

At a regional level, the northern region's average actual yield (Ya) was 1.05 t/ha and water-limited potential yield (Yw) was 2.66 t/ha, leaving a yield gap (Yg) of 1.61 t/ha and achieving a relative yield (Y%) of 42%. For the southern region, Ya was 0.82 t/ha and Yw was 3.20 t/ha, leaving a Yg of 2.38 t/ha and achieving a Y% of 25%. Interestingly, the southern region with its higher average Yw had the bigger yield gaps and achieved lower relative yields. This is not an unusual observation, but it is one that prompts us to ask why there are such large yield gaps for chickpea.

B. Paddock scale yield gap analysis

What did we do?

We used APSIM and local paddock data to conduct a paddock scale yield gap analysis on farmer fields in 2019, to see if we could determine the causes of any yield gaps found in the regional scale analysis. For this analysis, soil water and nutrients were measured by taking soil cores from a representative section of each paddock at sowing and harvest. Growers' paddock management data (e.g. row spacing, plant population, sowing date and depth) was also collected. This information was then used to conduct APSIM simulations of each field, in conjunction with local weather data. As with the regional scale analysis, APSIM was used to simulate yield that is attainable given growers' management practices and considering water availability (both rainfall and stored soil moisture) and other environmental conditions (e.g. temperature and solar radiation) through the season. APSIM simulations did not include the effect of nutrient deficiencies or of other biotic stresses on yield. We therefore expect the difference between observed and simulated yields to reflect a combination of simulation error, measurement error, biotic stress, and nutrient deficiencies.

After assessing the gap between paddock yields and simulated yields, APSIM was used to conduct 'what-if' scenario analysis of alternative management options for each field. This involved changing the APSIM simulations to use narrower rows (250 or 350 mm for southern and northern districts respectively), higher plant populations (40 plants/m^2) and earlier sowing (by up to 2 weeks where sowing had been delayed by rain). Simulated yield from the best 'what-if' scenario was compared to the originally simulated yield, to determine whether yield increases could be achieved by using these alternative agronomic practices.

What we found

When water availability is low, nutrient deficiency or diseases are unlikely to limit crop growth because water availability is the overriding limitation. In a dry year such as 2019, it was not surprising that on-farm yields of chickpea crops were generally close to simulated yields (Figure 6). On-farm yield in the monitored section of each paddock was usually within 300 kg/ha of the simulated yield, within the margin for error caused by variable soils across the monitoring transects. Out of the remaining six fields, three were only slightly outside the threshold and had some uncertainty about soil type and/or soil water at sowing below the sampling depth. However, the remaining three paddocks all had good levels of sowing soil water (110-150 mm) and may have been nutrient deficient (attainable yield gaps of 0.4, 0.6 and 1.3 t/ha). Two paddocks had low levels of P which would have been largely unavailable in the surface layer in a dry year. The other paddock had low sulphur levels with previous sulphur responses in canola and may also have been deficient in potassium because the soil type is naturally low in potassium, although this was not tested for this paddock.

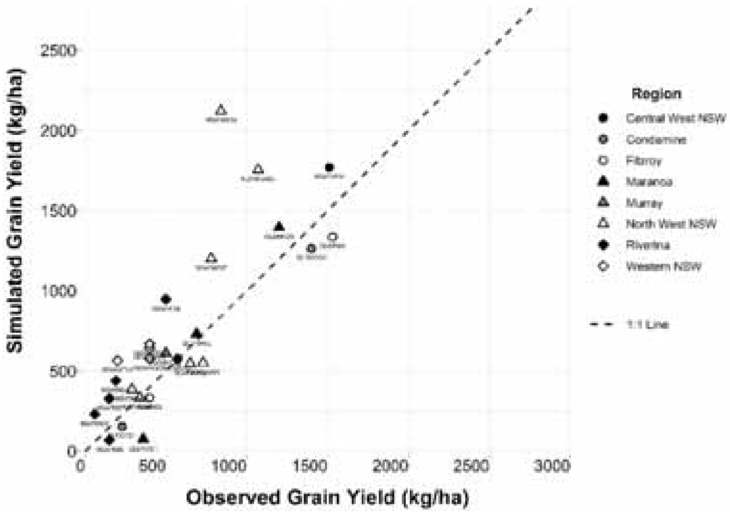


Figure 6. APSIM simulated vs measured yields for on-farm chickpea monitoring sites in 2019.

Although most paddocks did not appear to have significant nutrient deficiencies or disease problems, it is still possible that alternative agronomic practices may have led to yield gains in a below average rainfall season such as 2019. Our 'what-if' scenario analysis (Figure 7) showed that when using the predicted best management practices (i.e. optimum plant population, row spacing and sowing date) APSIM simulated grain yield was 0.4 t/ha higher on average across all the monitored fields. Paddocks with greater soil water at sowing showed the largest potential for improved yield, while extremely drought stressed fields gained little or no benefit from the alternative agronomic management. These simulated results should be tested further in field experiments.

One consideration yet to be resolved is whether increased plant populations should be used in combination with narrower row spacing, as changing both factors simultaneously may be necessary to realise the full yield benefit of either practice. An additional consideration is whether recently released chickpea cultivars can produce greater yield than older cultivars when sown early. Field trial data from our project shows that potential yield of chickpea can decline rapidly (up to 250 kg/ha per week that flowering is delayed (Figure8)), so ensuring chickpea crops flower at the optimum time is important for maximising grain yield.

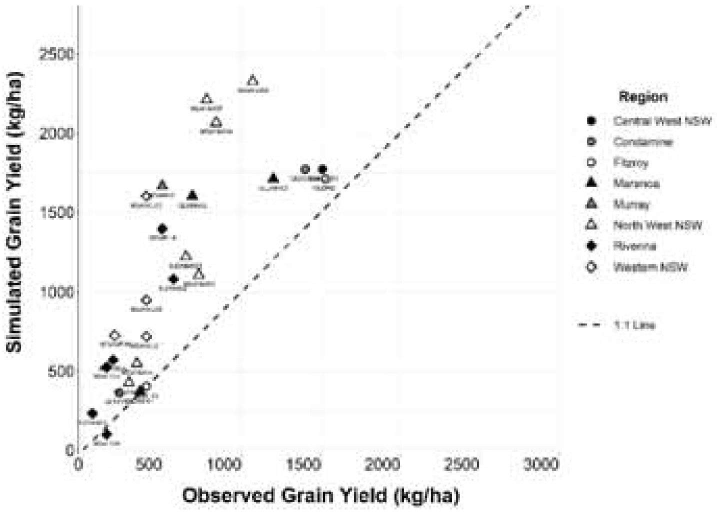


Figure 7. APSIM simulated vs measured grain yield for the best practice treatments discovered by ‘what-if’ scenario analysis of on-farm chickpea monitoring sites in 2019.

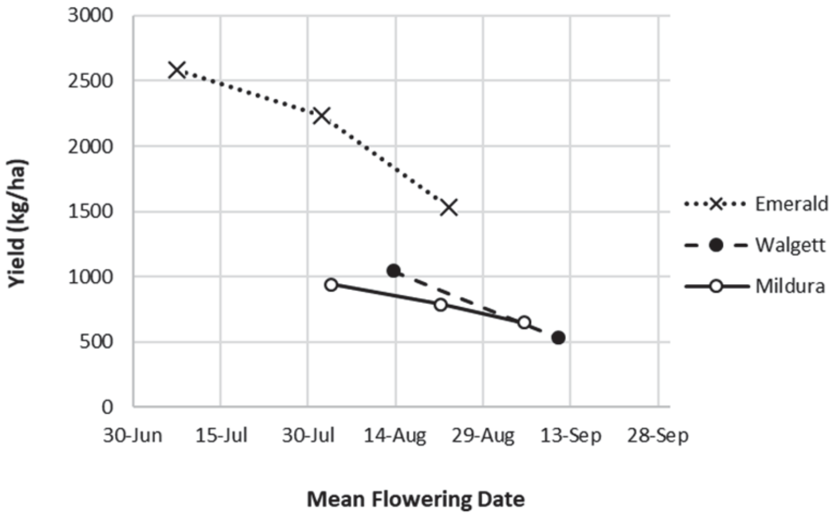


Figure 8. Average grain yield vs flowering date of 5 desi chickpea varieties from three ‘time-of-sowing’ field experiments conducted in 2019. Markers (cross or circles) represent average data generated from a single sowing date.

C. Determining optimum sowing date

What did we do?

A key aspect of addressing yield gaps is ensuring that crops are sown at a time that means they will flower when frost risk is declining but heat risk is still low. One of our project’s aims was to use APSIM to better understand when this ‘optimum flowering window’ occurs, and when should growers sow to achieve it. APSIM was therefore used to predict flowering date (measured as the date of appearance of the first flower on 50% of plants) and water-limited potential yield for the last 20 years (2000-2019), for a range of sowing dates (every fortnight from 1st March to 1st August) at 13 locations across the northern region, although we only present 6 from northern NSW in this paper. All simulations in northern NSW assumed that stored soil water on the 1st of March was 90 mm. Simulated row spacing was 40cm, sowing depth was 5cm and plant population was 30 plants/m^2.

Frost and heat-stress risk assessments were conducted by examining daily minimum and maximum temperatures with the potential for frost/heat stress (below +2°C and above +30°C at the standard met-station height of 1.2m). Accumulated degrees below/above these thresholds for a given time period (e.g. last week in August) was compared with the time periods that recorded the most severe frost conditions at Breeza and the worst heat-stress conditions at Emerald over the entire year. This created an estimate of relative risk that could be used to compare different production environments in assessing the flowering and sowing dates needed to avoid extreme temperature events. A function was developed within APSIM to account for chilling sensitivity of chickpea that prevents pollination and causes flower abortion at mean daily temperatures below 15°C (Clarke and Siddique, 2004) and delays pod-set, so the emphasis herein is on the risk of intermittent frost events that may cause yield-limiting damage to floral structures and developing pods.

We prepared charts (Figure 9,10) to estimate the sowing dates needed to ensure flowering occurs in the optimum window where frost risk is acceptable and water-limited yield potential is still high. For ease of presentation we have presented results for a single cultivar (PBA HatTrick⦶) which represents a medium maturity cultivar. If quicker/slower maturing cultivars are chosen, then sowing date should be adjusted to be slightly later or earlier to achieve the same flowering data. More data needs to be collected using the new phenology key (Whish et al., 2020) in order to better understand the comparative maturity of more chickpea cultivars across environments, but local knowledge can be used in the interim to decide whether cultivars will flower earlier or later than PBA HatTrick⦶.

Interpreting the potential yield graphs

In these graphs the box and whisker plots have a small black line which indicates the median yield. The yield from 50% of years fall inside the box (i.e. yields from 10 years, with five years either side of the median yield), and the box whiskers represent 1.5 times the value of the adjacent quartile (i.e. between the median and the edge of the box), while grey dots represent outliers beyond this range.

Determining optimal sowing date

A shaded box is used to connect three-graph sets from the same location showing the predicted optimal time of flowering and pod initiation. The beginning of this 'optimum flowering window' is determined by assuming that the earliest flower needs to appear in the last week when relative frost risk is still above 20% (approximately one day with a minimum temperature of 0°C in the week), as additional experiments suggest that the most sensitive growth stage to stress possibly falls 100 degree days later than the appearance of the first flower (Dreccer et al., 2020). The end of the optimum flowering window is the point when relative heat risk of 20% is reached, or when 5 weeks have elapsed since the window opened. An arrow has been inserted in the week where average temperature increases above 15 degrees (and the risk of cool temperature flower abortion is reduced), to help demonstrate when pod-set is likely to begin. In the middle graph for each location it is possible to determine which sowing dates can be used to achieve the optimal flowering dates, by referring across to the y-axis.

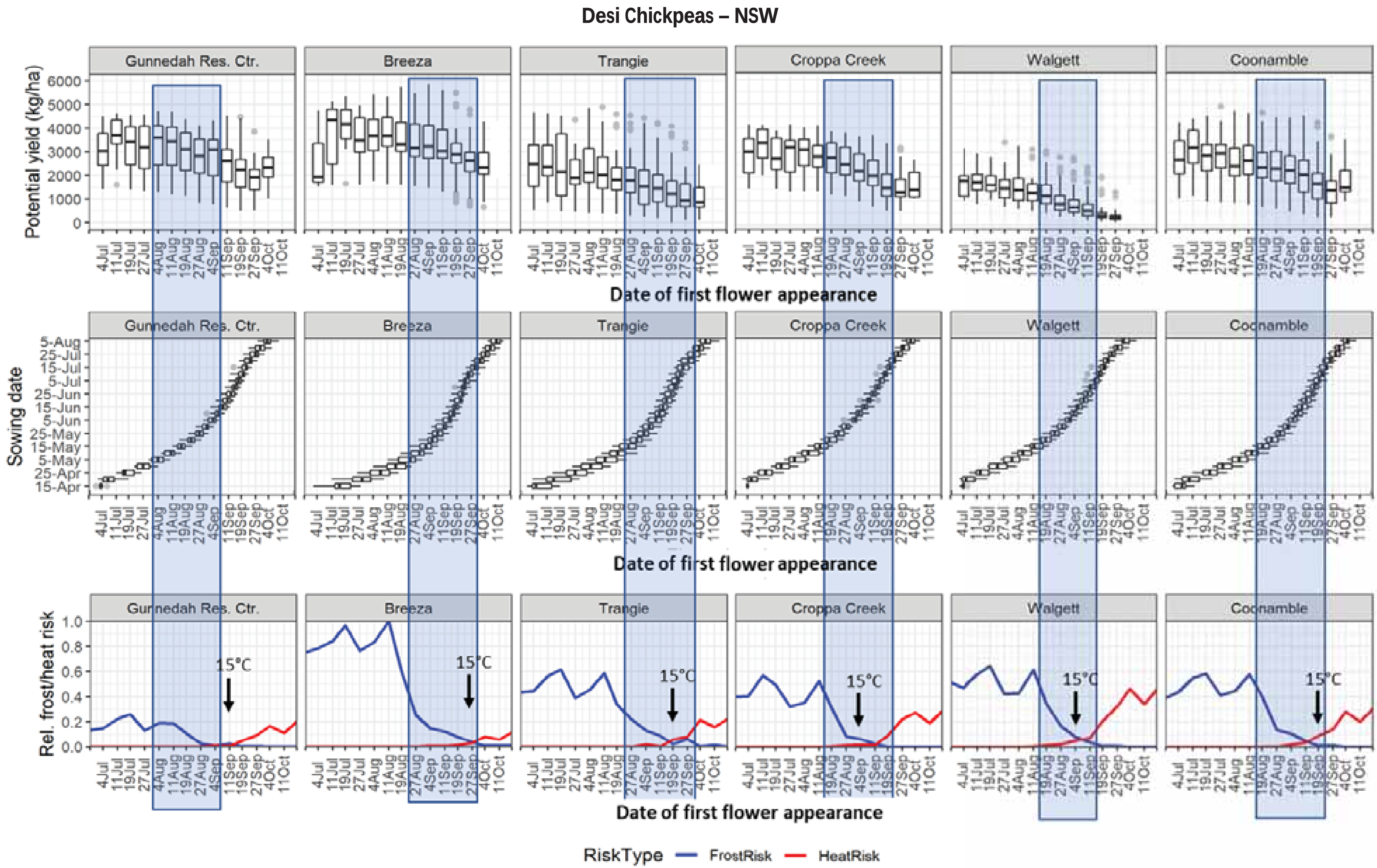


Figure 9. Water-limited yield potential, sowing date and relative frost/heat risk, plotted against the date of first flower appearance for PBA HatTrick⦶ at six locations in northern NSW. Shaded area represents the predicted optimum window for appearance of first flowers. Arrows and '15°C' mark the first week that has an average daily temperature greater than 15°C in the second half of the year, i.e. when conditions become favourable for pod-set.

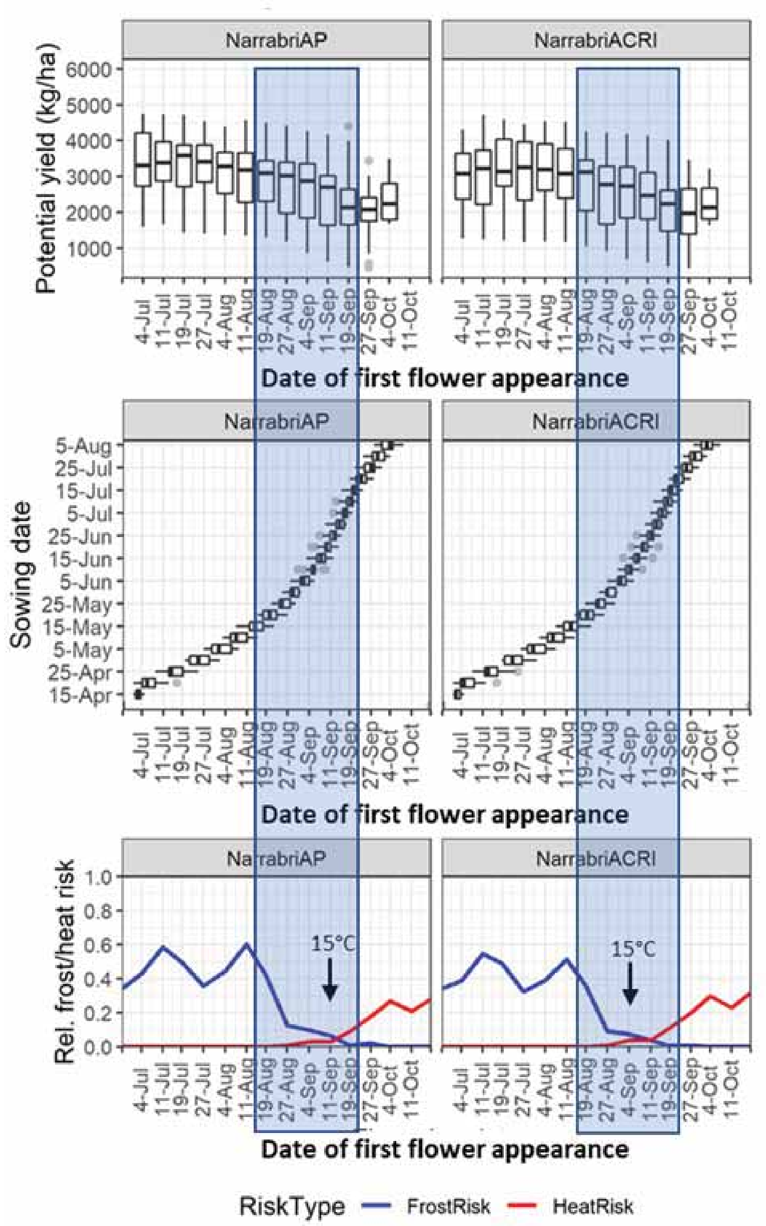


Figure 10. Water-limited yield potential, sowing date and relative frost/heat risk, plotted against the date of first flower appearance for PBA HatTrick⦶ at Narrabri Airport and the Australian Cotton Research Institute (ACRI). Shaded area represents the predicted optimum window for appearance of first flowers. Arrows and '15°C' mark the first week that has an average daily temperature greater than 15°C in the second half of the year, i.e. when conditions become favourable for pod-set.

Trends and observations

Water-limited potential yield declines rapidly in spring with later sown crops, particularly in the more northern environments.

Within the optimum flowering window, yield declined up to 300 kg/ha per week that first flower appearance was delayed in the most extreme situations (e.g. Croppa Creek (Figure 9)). Although the model showed a tendency to over-predict grain yield on early sowing dates in some environments (Figure 11), yield was lower for later sowing dates in the field trials (Figure 8), where grain yield declined at between 60 and 225 kg/ha per week that appearance of the first flower was delayed, depending on location and time-of-sowing. While it can be difficult to determine if yield reductions are due to heat stress, water stress or simply the shorter growing season experienced by late sown crops, delayed flowering into warmer weather conditions reduced yield in the 2019 field trials and the simulations. Nevertheless, further model testing is necessary in different seasons to ensure that the model can successfully simulate different types of season (e.g. early stress followed by mid or late-season rainfall). In wet seasons delayed sowing may not have the same yield penalty, so further research is necessary to investigate the relationship between time of sowing and yield across a range of locations and seasons. It should also be remembered that simulations were prepared using 90mm of stored soil water prior to sowing. Water-limited yield potential will vary from that presented here if more/less water is available at sowing.

It is unclear whether ultra-early sowing could provide even greater yield advantages in a dry season

While most northern sites reached the 15°C threshold for pod set 2-3 weeks after the beginning of the optimum flowering window, the more southern sites had a larger gap (e.g. 5 weeks at Wagga and 9 weeks at Mildura: data not shown). While early sown crops may not set pods for many weeks after the first flower appears, the increased biomass development and/or root growth that occurs during this delay may be important for maximising grain yield in a dry season. While Richards et al. (2020) identified a yield penalty associated with ultra-early sowing in southern NSW, their results were obtained in different environments to those of southern QLD and northern NSW. Further research is recommended in both high and low frost-risk environments with a range of stored soil water to understand how long this infertile flowering phase can continue before significant yield loss occurs. This research would help define the optimum flowering window and sowing dates with more precision for a range of environments and seasons.

Frost risk varies over short distances within the landscape, so individual farms or paddocks may have different risk profiles (and optimum sowing dates) to those presented here.

Recent work by AMPs Research in wheat has shown that the top sections of some sloping paddocks on the Liverpool Plains can be sown 4-6 weeks earlier due to lower frost risk. Similarly, it is important to understand that the charts presented here may not be applicable to nearby farms or paddocks due to differences in local topography. For example, our first simulations for the Liverpool Plains used the Gunnedah Resource Centre meteorological station, but we later realised this data was not representative of most farms in the Gunnedah district. We then compared these simulations with the equivalent data from nearby Breeza (side-by-side comparison in Figure 9) showing a much greater frost risk at Breeza compared to the Resource Centre for the same dates in August. Growers should seek independent advice on frost risk for their individual locality, and be careful about making large changes to their normal sowing dates based on data presented here.

Kabuli cultivars are generally slower maturing than desi cultivars

Growers swapping between the two types for the first time need to be mindful of sowing Kabuli chickpeas earlier than desi chickpeas, to flower in the same optimal flowering window.

Extremely early and ultra-late sown crops have the lowest water-limited potential yield.

Some growers in southern NSW are experimenting with spring-sown chickpea to avoid the overly wet, winter growing conditions they encounter in good seasons. However, they may struggle to generate acceptable grain yield unless they are in a cool environment and/or have a genuinely full profile of moisture, and experience good spring rainfall. Further simulations could be conducted to investigate these possibilities.

When testing APSIM on 2019 field trials not discussed in this paper, APSIM simulated the maximum grain yield for field trial data reasonably well, although grain yield for the earliest time of sowing was overpredicted at most locations except Emerald (data not shown). This may have occurred because the effects of cold temperature or frost on flower viability or pod-set are not being fully simulated. Alternatively, the first time of sowing typically has the highest potential yield, so any nutritional limitations are also more likely to impact grain yield in the early time of sowing. Further research is necessary to determine the reasons for these yield gaps.

An example of this is shown in Figure 11 where PBA HatTrick⫯ had similar field-measured grain yield across sowing dates and irrigation treatments at Greenethorpe, but potential (simulated) yield of the irrigated early sown treatments was much higher than measured grain yield, causing a large 'yield gap'. In this experiment PBA Slasher⫯ was much closer to achieving potential yield across sowing dates under irrigation and achieved 50% more grain yield in the irrigated trial on the first time of sowing compared to the third time of sowing, but still had a sizeable yield gap of 1 t/ha on the early sowing date. Further research is needed to determine if cultivar choice or other agronomic practices can be used to improve grain yield in conjunction with early sowing dates under favourable conditions.

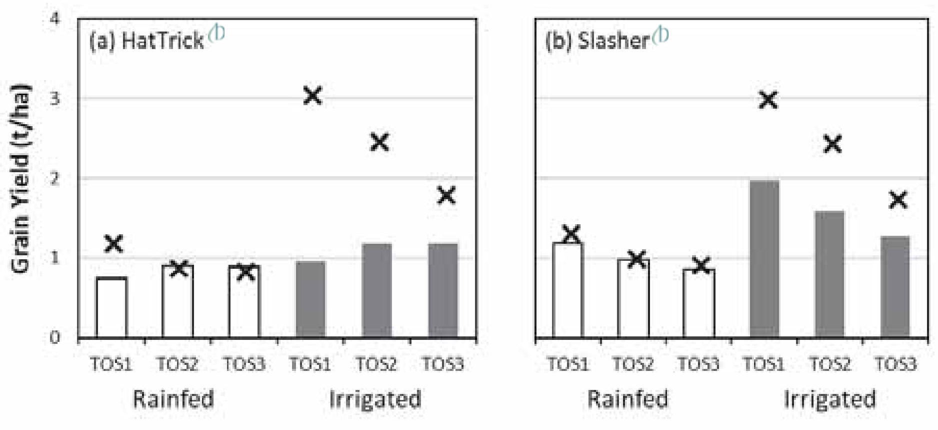


Figure 11. Measured grain yield (bars) and APSIM simulated yield (crosses) for the desi cultivars (a) PBA HatTrick⫯ and (b) PBA Slasher⫯ across three times of sowing (TOS) at Greenethorpe, 2019. Sowing dates were 30-April, 21-May, 12-Jun. Standard error is ≤ 0.1 t/ha for all measured yield data

Effect of deep sowing and water stress on optimum sowing date

As discussed above, the predictions of optimum sowing date in Figures 9-10 assume a sowing depth of 5cm. As chickpea is often sown deeper than this, it is possible growers may need to take this into account when deep sowing, by sowing earlier than the sowing dates presented here to achieve the same flowering date. However, experiments in Western Australia on sandy soils have shown negligible differences for emergence date and flowering date for an early-season chickpea experiment sown at 5 and 20cm (Rich and Lawes, 2020). Additional anecdotal evidence suggests that while the delay may be small in a warm environment (e.g. Central Qld), it may be larger in cooler environments such as the Liverpool Plains. A rule of thumb used by some agronomists is to allow one days delay in emergence for every additional centimetre of sowing depth, however it is possible that deep sown seedlings may be accumulating 'heat units' toward their phenological development while underground, so delayed emergence may not lead to an equivalent delay in the appearance of the first flower. More research is necessary to determine the effect of sowing depth on emergence date and phenology processes across QLD and NSW pulse production regions. Growers and agronomists are advised to keep records on this information for individual fields, as it probably varies between soil types and location.

Additional experiments from 2019 also showed that flowering date sometimes occurred earlier in rainfed plots compared to irrigated plots, suggesting that water stress may hasten reproductive processes. The trend was not consistent across locations and cultivars, but in the most extreme case (Genesis 90 at Greenethorpe) flowering processes were 10-19 days earlier in rainfed treatments compared to irrigated treatments, while at Gatton no differences were detected in flowering date for any cultivar but podding date was 4-12 days earlier in rainfed treatments of both PBA HatTrick⟑ and Genesis™ 90. The degree of water stress was probably different between these locations, which would explain the variable results. Further work is needed to determine how different severity of water stress might impact on the occurrence of the optimal flowering window for specific situations and soil types. Wet conditions might reasonably be expected to delay flowering (and hence require earlier sowing to achieve the same flowering date) as has been observed in crops such as wheat. However, disease control may be more difficult in conjunction with early sowing in wet seasons, so optimum sowing date from a grower's perspective may ultimately not differ greatly between wet and dry years.

Conclusions

While acknowledging the uncertainty in calculation of yield gaps for chickpea crops, and the need for further work on chickpea model development as well as more systematic chickpea farm data collection, we are confident that the gaps described are broadly realistic if not accurate. Other research to identify the causes of yield gaps in Australia's cropping zone have identified that for wheat, nitrogen was the single most limiting factor. This is less likely to be the case for legume crops such as chickpea that can meet their nitrogen requirement by symbiotic N fixation, although some anecdotal evidence suggests it could be an issue on certain soil types in high-rainfall seasons or when residual chemicals affect rhizobium activity.

The process of investigating experiments using the models also provides direction for future research by highlighting the scientific 'bottlenecks' that improved prediction and understanding of crop performance. Additional research is needed across additional seasons to ensure that the predictions of crop performance apply to a range of seasonal conditions, not just the extreme drought conditions experienced during 2019. Model testing in additional environments and seasons will undoubtedly highlight further strategic research priorities that could be addressed to benefit Australian grain farmers into the future.

While Australian grain producers are among the most efficient in the world, there is still room for closing the yield gap of chickpea crops even in a drought year, by adopting best agronomic practice while aiming for a maximum profit level adjusted for individual growers' risk appetite.

Acknowledgements

The research was undertaken as part of this project is made possible by the support of the GRDC Project 'The adaptation of pulse (chickpea and lentil) across the northern grains region' (CSP1904-005RXT). We also acknowledge the many growers, agronomists and researchers who assisted by providing paddock monitoring data and conducting the time of sowing trials, including Michael Moodie (Frontier Farming Systems), Matt Gardner (AMPS Research), Doug Sands and Peter Agius (DAFQ), Jeremy Dawson (MCA Agronomy), Peter Baird (Dodgshun Medlin), Kate Coffey (Riverine Plains Inc), Barry Haskins and Albert Gorman (Ag Grow Agronomy and Research), Chris Minehan and Fred Broughton (Rural Management Strategies), and Jim Hunt (Hunt Ag).

CSIRO, NSWDPI, SARDI, QDAF, USYD, UQ-QAAFI, The Australian Federal Government, QLD, NSW and SA State Governments are also acknowledged for directly and indirectly contributing to project funding. We would also like to thank Jeremy Whish, Fernanda Dreccer and Tony Swan (CSIRO), Richard McCallum, Mark Richards and Kirsty Hobson (NSWDPI), Helen Bramley and Josie Clarke

(University of Sydney) for their contributions to the project that were not reported in this paper. We thank Paul McIntosh (Pulse Australia and the Australian Herbicide Resistance Initiative) for assisting with disease control protocols of field trials, and Neil Huth (CSIRO) for assisting with APSIM model development. We also acknowledge the incredible work carried out by the many technical staff from many organisations in successfully planning and conducting the extensive field trial program, which was instigated at short notice.

References

Clarke, H. J. and Siddique, K. H. M. (2004). Response of chickpea genotypes to low temperature stress during reproductive development. *Field Crops Research*, *90*, 323-334.

Dreccer, M.F., Sadras, V., Lake, L., Peake, A.S., Lee Long, I. et al. (2020). Appendix B: Understanding the critical stage for yield formation in chickpea and lentil in response to abiotic stress. *In* Peake A.S. et al. (Eds) 'Final Report to GRDC project CSP1904-005RXT: The adaptation of pulses (chickpea and lentil) across the northern grains region'. CSIRO Agriculture and Food, ACT, Australia.

Lobell, D. B., Cassman, K. G., & Field, C. B. (2009). Crop yield gaps: their importance, magnitudes, and causes. *Annual Review of Environment and Resources*, *34*

Rich, S. and Lawes R. (2020) Sowing flexibility of chickpea and lentil in the Western Australian farming system. GRDC Updates, February 2020, Perth.

Richards, M., Maphosa, L., Presto, A., Napier, T., Hume, I. (2020). Pulse adaptation – optimising grain yield of chickpea and lentils. GRDC Northern Region Updates, 2019. Accessed online at: https://grdc.com.au/resources-and-publications/grdc-update-papers/tab-content/grdc-update-papers/2020/02/pulse-adaptation-optimising-grain-yield-of-chickpea-and-lentils

van Rees, H. McClelland, T. Hochman, Z. et al. 2014. Leading farmers in South East Australia have closed the exploitable wheat yield gap: Prospects for further improvement. *Field Crops Research*, 164, 1-11.

Whish, J., Peake, A.S., Rodriguez, D., Swan, T., Richards, M., et al. (2020). Appendix F: Chickpea Phenological Key. *In* Peake A.S. et al. (Eds) 'Final Report to GRDC project CSP1904-005RXT: The adaptation of pulses (chickpea and lentil) across the northern grains region'. CSIRO Agriculture and Food, ACT, Australia.

Contact details

Allan Peake
CSIRO Agriculture and Food
Ph: 07 4571 3212
Email: allan.peake@csiro.au

Zvi Hochman
CSIRO Agriculture and Food
Ph: 07 3214 2234
Email: zvi.hochman@csiro.au

Important disclaimer

consequences, including but not limited to all losses, damages, costs, expenses and any other compensation, arising directly or indirectly from using this publication (in part or in whole) and any information or material contained in it.

Viral diseases in faba bean, chickpeas, lentil and lupins. Impacts, vectors/causes and management strategies for 2021

Joop van Leur and Zorica Duric
(NSW DPI, Tamworth Agricultural Research Institute)

Key words

faba bean viruses, *Bean yellow mosaic virus*, *Alfalfa mosaic* virus, aphids

GRDC codes

DAN00202, DAN00213/BLG209, DAN00213/BLG204

Take home messages

- The severe virus epidemic in faba bean in northern NSW during 2020 was initiated by early and massive flights of aphids (mainly cowpea aphids) that carried *Bean yellow mosaic virus* (BYMV) into the crops
- After a 2-year drought, heavy January and February rains in north-west NSW triggered the emergence of naturalised medics and other pasture legumes, which allowed a build-up of aphids and virus prior to the emergence of faba bean crops
- Most faba bean crops were sown into bare ground as cereal stubble was lacking after the extended drought. Lack of standing cereal stubble or uneven emergence makes pulse crops particularly vulnerable to early aphid infections
- Relatively mild and dry conditions during the start of the season favoured aphid multiplication in the faba bean crops and a fast spread of virus from initial infection foci
- Co-infections of BYMV and *Alfalfa mosaic virus* (AMV) caused particularly severe symptoms in several crops
- Both BYMV and AMV are non-persistently transmitted viruses that require only a short probing period by a viruliferous aphid to infect a plant. Slow-acting insecticides like imidacloprid applied to seed will not prevent infection of non-persistently transmitted viruses by incoming, winged, aphids . However, they may help in slowing the multiplication of aphids in the crop and subsequent spread of the virus by wingless aphids
- Further work is needed to investigate how long an imidacloprid seed dressing remains active and whether a foliar application of a pulse registered product (pirimicarb) after aphids are found in a crop is more practical, economic or effective
- There are no indications that transmission of BYMV to faba bean seed occurs at significant levels
- Virus control strategies are all based on preventing infection, particularly during the early growth stages of the crop. Poorly emerging crops or crops sown into bare ground are particularly vulnerable to infection. Sowing in standing cereal stubble and using high quality seed with good seedling vigour has shown to be the most reliable management option to avoid virus infections.

Pulse viruses and pulse virus vectors

Pulses are far more vulnerable to virus infection than winter cereals with over 20 viruses considered to be of economic importance on pulse crops worldwide. All major pulse viruses require an insect vector (mostly an aphid species) to transmit the virus to a healthy plant. The mode of transmission can be non-persistent, which means that the aphid only needs a short probe of the plant in order to acquire or to transmit the virus, while persistently transmitted viruses require a longer feeding period by the aphid. Plants can be infected by most non-persistently transmitted viruses under experimental conditions through rubbing a virus suspension into the leaf ('mechanical inoculation'), while a viruliferous vector is always needed for the persistently transmitted viruses. Viruliferous aphids will lose a non-persistently transmitted virus after probing a healthy plant, but aphids that carry a persistently transmitted virus remain viruliferous.

The difference in the time needed to transmit a virus has implications for the effectiveness of aphicides to control virus infections; infection by a persistently transmitted virus like *Bean leafroll virus* (BLRV) can be prevented by a timely foliar application of an insecticide or by seed dressing with a systemic insecticide, but most aphicides are not fast enough to prevent infection by a non-persistently transmitted virus like *Bean yellow mosaic virus* (BYMV).

Virus epidemiology and the prediction of severe virus epidemics is complicated as aphid numbers and movements depend on numerous factors, both during and preceding the growing season. A few viruses can survive in seed, but most require a 'green bridge' of living host to survive between cropping seasons. Pulse viruses are not host species specific and naturalised pasture legumes, like medics or sub clovers, or perennial legumes, like lucerne or white clover, can be infected with a range of viruses that can be harmful to grain legumes like faba bean or chickpea. Viruses like BYMV and AMV can be seed transmitted in several pasture legume species and viruses and their vectors can build up in pastures, roadside weeds or stock routes prior to the emergence of pulse crops. Also, a virus that can devastate grain legumes might have little or no impact on a pasture legume or a weed; e.g. *Bean leafroll virus* (BLRV) kills pulses like faba bean, field pea, lentil and field pea, but is symptomless on lucerne, its main summer host.

Different aphid species have different temperature optima, but generally mild weather favours aphid multiplication. Aphids can multiply very fast and host plant population can become rapidly overcrowded, particularly if the host plants are still in an early stage of development (as with naturalised pasture legumes during late summer-early autumn). Overcrowding or deterioration of the host plant will trigger the development of winged aphids who can, largely through wind currents, migrate over long distances. Large peaks in movements of winged aphids can be observed during autumn and can be harmful for early sown crops like faba bean and lupins. Aphid activity generally slows down during winter but picks up again with rising temperatures and major aphid flights can be again observed in spring. Later-sown pulse crops like chickpeas generally escape the autumn aphid flights but are vulnerable to the spring flights.

Faba bean viruses in northern NSW

Within Australia 12 viruses have been reported to infect faba bean, but not more than five have caused serious yield reductions in the past (Table 1). Surveys in northern NSW over the last two decades identified *Bean leafroll virus* (BLRV) as the most important faba bean virus. Severe symptoms, like stunting and plant death, were found after infection by BLRV and (less frequently) by the closely related *Soybean dwarf virus* (SbDV). Heavy yield losses by BLRV were recorded in the early 2000's on the highly susceptible cultivar Fiord, particularly in paddocks close to lucerne, the major BLRV summer host. Germplasm screening for resistance was successful in identifying good sources of BLRV resistance, which were used in the breeding program to develop varieties with improved resistance.

In 2004 and 2005 high incidences of the thrips transmitted *Tomato spotted wilt virus* (TSWV) were found in faba bean paddocks north of Moree. The sudden occurrence of TSWV was likely a result of the recent incursion of the western flower thrips (*Frankliniella occidentalis*), a highly effective vector. TSWV has a very wide host range and is lethal on faba bean. However, although infections of up to 10% were found in some crops, infected plants remained randomly scattered through paddocks and, surprisingly, no secondary spread could be noticed. TSWV was rarely found in more recent surveys. Other viruses that can cause plant death, but only occur sporadically, are *Sub clover stunt virus* (SCSV) and *Clover yellow vein virus* (CYVV).

Table 1. Faba bean viruses in Australia (in alphabetical order)

Virus	Abr.	Transmission	Major vectors	Seed transmission[1]	Occurrence in NSW faba bean crops	Impact on plants
Alfalfa mosaic	AMV	non-persistent	many aphid species	medics, lucerne, lupin	low incidences every year	moderate
Bean leafroll	BLRV	persistent	pea, cowpea aphid	no	very high occasionally	high
Bean yellow mosaic	BYMV	non-persistent	many aphid species	medics, sub clover, (faba bean)	high incidences regularly, normally at end of season	high after early infection
Broad bean wilt	BBWV	non-persistent	green peach aphid and others	no	very rare	high
Cucumber mosaic	CMV	non-persistent	many aphid species	lupin, lentil	very rare	low
Clover yellow vein	CYVV	non-persistent	cowpea aphid	no	rare	high
Pea seed-borne mosaic	PSbMV	non-persistent	many aphid species	field pea, (lentil)	rare	low
Phasey bean mild yellows	PBMYV	persistent	cowpea aphid	no	?	?
Sub clover stunt	SCSV	persistent	cowpea aphid	no	generally rare, widespread occasionally	high
Soybean dwarf	SbDV	persistent	pea aphid and others	no	low incidences regularly	high
Turnip yellows[2]	TuYV	persistent	green peach aphid	no	high incidences regularly at end of the season	moderate
Tomato spotted wilt	TSWV	persistent	Western flower thrips and other thrips	no	occasionally low incidences	high

[1] Seed transmission of host species listed between brackets is reported, but unlikely to be of significance

[2] TuYV was previously known as *Beet western yellows virus* (BWYV)

While survey results indicated that BLRV was the most important virus because of its impact on yield, BYMV was always the most frequently identified virus throughout northern NSW. Generally, infections appeared late in the season and symptoms consisted only of a relatively mild mosaic in the top leaves above the pod setting nodes and no serious impact on yield was noticeable in commercial crops. However, yield loss trials comparing BYMV inoculated with non-inoculated plots (using mechanical inoculation at the 3-4 leaf stage) demonstrated that early infections could have a major impact on yield with over 50% yield reduction on PBA Warda⍦ and PBA Nasma⍦ (the two most common varieties in northern NSW) and close to 70% yield reduction on the highly susceptible southern variety Fiesta VF. Large numbers of breeding lines and germplasm accessions were evaluated in BYMV inoculated screening trials over the years, but no lines with high levels of resistance were identified. Unlike with BLRV, no plant death was observed in the yield loss or resistance screening trials, except for several germplasm accessions from Ecuadorian origin that showed hyper-sensitive reactions.

The 2020 faba bean virus epidemic in northern NSW

By early June 2020 serious symptoms in early sown faba bean crops were reported in several sites in northern NSW. Patches of necrotic and stunted plants in the affected paddocks resembled severe early infections by BLRV, SCSV or TSWV seen in earlier years. However, testing of symptomatic plants by Tissue Blot Immunoassay (TBIA) at the Tamworth Agricultural Institute (TAI) and by molecular tests at the QDAF virology laboratory in Brisbane (Dr Murray Sharman) showed the main virus to be BYMV, with co-infection by AMV in some paddocks (Table 2).

Table 2. Presence of three major faba bean viruses and one virus group in collected and submitted faba bean samples, northern NSW, 2020 [1]

Symptom type	Tested plants	% positive plants as determined by TBIA			
		BYMV	AMV	BLRV	Luteovirus[2]
Randomly collected	359	44.3	13.1	1.1	7.5
No symptoms	407	49.9	6.4	0.2	2.0
Not specified virus symptoms	279	58.4	15.4	2.5	9.0
Plant necrosis	329	81.8	26.4	0.3	2.4
Plant stunting	194	66.0	5.2	12.9	7.2
Leaf mosaic	234	88.9	11.5	0.0	3.8
Total plants tested	1802	62.7	13.3	2.1	5.0

[1] Minor incidences found for SCSV (7 plants out of 325 tested), CYVV (1 plant out of 991 tested) and *Cucumber mosaic virus* (CMV, 4 plants out of 682 tested)
[2] Luteoviruses other than BLRV

The early infections followed high populations of migrating aphids observed on aphid traps during autumn 2020 (Figure 1). The main aphid species found on the traps was cowpea aphid (*Aphis craccivora*), but other aphid species known to colonise pulse species were also found in high numbers: pea aphid (*Acyrthosiphon pisum*), blue-green aphid (*Acyrthosiphon pisum*) and green-peach aphid (*Myzus persicae*). Cowpea aphid is a highly effective vector of both persistently and non-persistently transmitted viruses, and - equally important - it colonises and multiplies on faba bean. Further surveys and testing during the season showed clear foci spreading from early infected plants, a pattern typical of colonising, wingless aphids.

Several agronomists expressed concern that green mirids (*Creontiades dilutes*), which were in abundance in faba bean crops during 2020, were capable of transmitting BYMV. We have undertaken two separate trials in which green mirids were left for 24 hours on BYMV infected faba bean plants in insect proof tents, before healthy faba bean plants were placed in the same tent. No

BYMV transmission from the infected to the healthy plants was observed, confirming numerous publications that only aphids can transmit this virus.

As the season progressed the virus spread through most faba bean paddocks in northern NSW, but severe symptoms generally remained restricted to early infected paddocks. Large differences in severity could be observed between paddocks that were at close distance, and paddocks close to the Queensland border and those south of Gilgandra were less affected (Figure 2). Virologists from QDAF confirmed the presence of BYMV in faba bean paddocks in Queensland, but severe symptoms were rare except for an irrigated crop near St George.

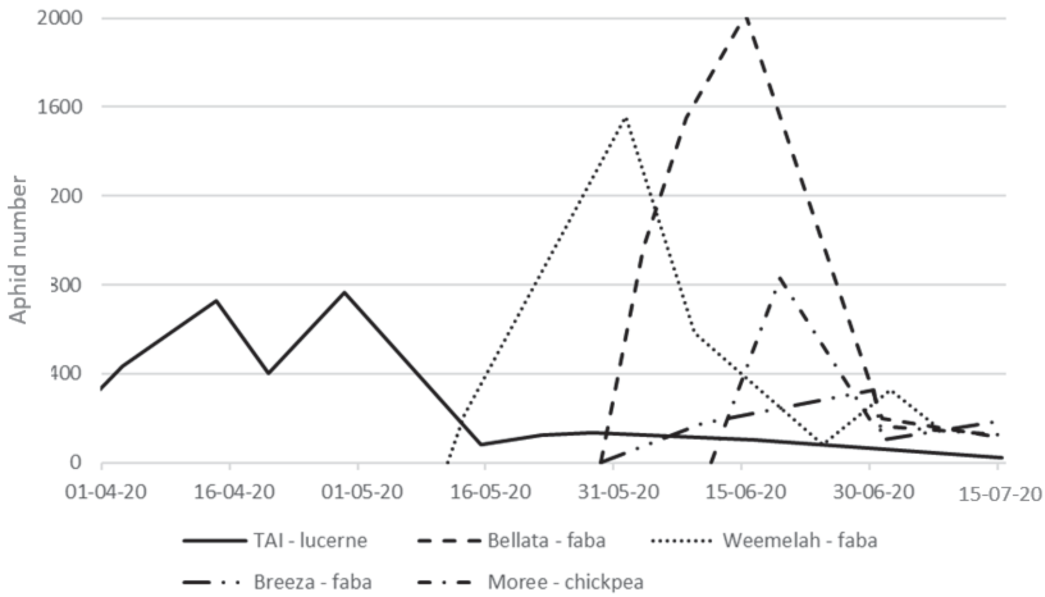


Figure 1. Aphids (number of aphids / m^2 yellow sticky trap / day) caught on yellow sticky traps in 5 sites in northern NSW, Autumn 2020

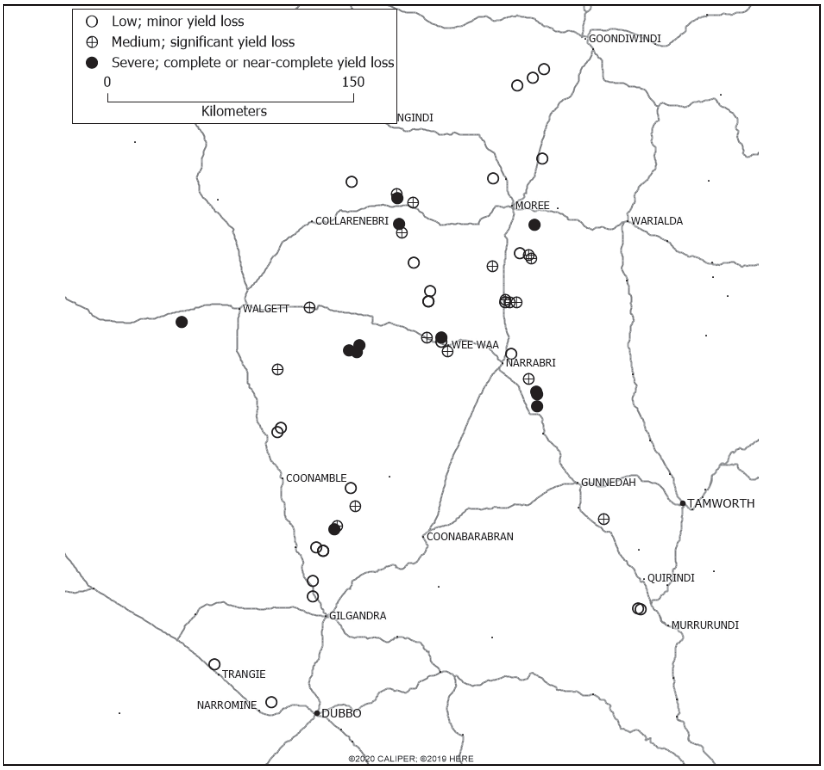


Figure 2. Severity of virus infections in 55 faba bean paddocks Northern NSW, 2020

The very high early aphid numbers and severe virus in north-western NSW points to a possible role of naturalised pasture legumes in the epidemiology of pulse viruses: After two very dry years, the January - February rains in the north-western part of the state would have triggered the emergence of large areas of medics and other potential virus hosts. Mild temperatures promoted a fast multiplication of aphids and when aphid populations on the medics became too crowded, winged aphids would have moved to newly emerged faba bean crops. BYMV (and AMV) are both transmitted in medic seed, so even with low seed transmission rates enough aphids could have picked up inoculum to establish infection foci in faba bean crops. Continuing mild temperatures during the start of the season allowed multiplication of aphids in the faba bean crops and spread of the virus from the initial infection foci. The preceding drought also had indirect effects on the high level of virus infections: Aphids tend to avoid crops sown in standing cereal stubble and with less cereal crops grown in 2019, more faba bean crops were sown into bare ground. Also, faba bean seed was in short supply and some seed used was rather old and less vigorous.

While the unusual severity of BYMV infection during 2020 was likely a direct result of very early infections and, in some cases, co-infection by AMV, it is also possible that more virulent BYMV strains were present. BYMV is known to be variable in pathogenicity and host species specificity. We have isolated a large number of strains this year and are in the process of evaluating these using pathogenicity testing and molecular tools.

Viruses in crops other than faba bean

Virus symptoms were observed in several chickpea crops in the Moree-Narrabri region that were bordering faba bean paddocks. The level of infection was high close to the border but diminished rapidly further from the faba bean crop. Testing of virus symptomatic plants showed that while the faba bean paddocks had high BYMV and minor AMV infection levels, symptomatic chickpeas were only infected by AMV. BYMV is reported to be capable of infecting chickpeas, but in our greenhouse tests chickpeas reacted as immune to the majority of BYMV strains isolated from faba bean.

Random samples taken from chickpea paddocks by Dr Kevin Moore as part of his yearly disease survey (Table 3), showed that the later sown chickpea crops generally escaped the aphid autumn flights. The absence of virus spread in chickpea paddocks also demonstrated that aphids rarely colonise chickpeas and infection is generally caused by incoming, winged aphids.

Table 3. Presence of four major pulse viruses and one virus group in collected and submitted samples of pulses (other than faba bean), NSW, 2020.

Species	Tested plants	% positive plants as determined by TBIA				
		BYMV	AMV	BLRV	Luteovirus[1)]	CMV
Chickpeas	1,329	0.0	5.5	0.3	1.1	1.1
Lentils	1,161	0.2	12.7	2.7	0.3	34.6
Lupins	227	1.3	28.6	0.9	0.0	90.3

[1] Luteoviruses other than BLRV

Both lupins and lentils are, unlike chickpeas, crops colonised by several aphid species and highly vulnerable to infection by a range of viruses. CMV caused severe symptoms in narrow-leafed lupin crops in the Gilgandra region with co-infection by AMV aggravating losses in several paddocks. CMV can be seed-transmitted in narrow-leafed lupins at high rates and testing of several seed lots that were used for the 2020 sowing showed dangerously high CMV levels. Narrow-leafed lupins in NSW are generally used as stockfeed on farm and most growers multiply their own seed stock for several years. While virus infections might remain unnoticed in most years, seed infection levels can build up slowly and cause a severe virus epidemic in an aphid favourable season like 2020.

Lentils are still an experimental crop in most of NSW, but the high levels of CMV and other viruses in lentil samples taken from trial sites (Table 3) showed that viruses may become a major factor limiting the expansion of lentils.

Virus control strategies

Virus control strategies will differ between crops and virus species, but all are aimed at avoiding infection as curative control of viruses is not possible.

Minimising numbers of incoming virus vectors

The most effective strategy currently is to minimise virus infection by promoting fast canopy closure through following optimal agronomic practices and use of high quality seed with good seedling vigour. Migrating aphids prefer landing in crops that show uneven emergence and bare ground, while sowing in standing stubble deters aphid landings. Early sowing increases the risk of virus infection as crops are exposed to the autumn aphid flights when plants are small and most vulnerable to virus infection. However, later sowing is often not a practical option for growers.

Virus-free seed

Several non-persistently transmitted viruses can be seed-borne in pulses. Sowing virus-infected seed will result in infection foci scattered randomly throughout a crop right after crop emergence and a rapid spread of the virus. Fortunately, for most virus / pulse host combinations, the levels of seed transmission are very low and, while still very important for quarantine, not of concern in commercial crops. The exceptions are CMV in narrow-leafed lupins and lentils and *Pea seed-borne mosaic virus* (PSbMV) in field peas. Growers who keep their own seed should have it tested and only use virus-free seed.

BYMV can be seed-transmitted in faba bean, but so far we haven't found any evidence of its presence in commercial or farmers' seed lots. However, new virus strains can develop that have a better ability to be seed transmitted, so our testing program is continuing.

Avoiding inoculum sources

Unlike most fungal pathogens, viruses don't survive in stubble or soil. Apart from those that are seed-transmitted all will be brought into the crop by vectors from outside sources. It is advisable to keep distance from known sources of infection like lucerne. Weed and volunteer legume crops within or near crops are also key virus sources and should be controlled before the crop emerges.

Chemical control of vectors

The effectiveness of aphicides to control virus infection differs between viruses. Viruses that are persistently transmitted, like BLRV or TuYV, require a relatively long feeding period and a seed treatment with a systemic insecticide can provide protection during early growth when plants are most vulnerable. Non-persistently transmitted viruses, like BYMV, CMV or AMV, only require a brief probing to transmit virus and the insecticides used for seed treatment act too slowly to prevent infection.

Imidacloprid is registered as a seed treatment for early aphid control on faba bean, field pea and lentils. The seed treatment will not stop initial BYMV infections from migrating aphids, but it could possibly delay the build-up of colonising aphids in a crop and thereby limit virus spread. Greenhouse trials in 2020 confirmed that imidacloprid seed treatment reduced the multiplication of cowpea and pea aphid numbers on faba bean, but it took more than one day to kill aphids. Field research is needed to determine how long an imidacloprid seed treatment remains effective and whether it will be more useful than a well-timed foliar aphicide application.

Resistance

Genetic resistance can be a very economical and environmentally friendly option for virus control. Over the years good resistance has been identified for several pulse / virus combinations and subsequently used in Australian pulse breeding programs.

Unfortunately, the search for high levels of BYMV resistance in faba bean has not been successful so far. There are differences among Australian faba bean varieties in symptom expression with some of the older varieties and lines developed for the southern region reacting as 'very susceptible'. There were no indications that the current northern region varieties differed in their BYMV reaction during the 2020 epidemic, but a few breeding lines yielded well in different sites under severe BYMV pressure. These lines will be tested during the 2021 season in inoculated trials.

All narrow-leafed lupin varieties are CMV susceptible, but a number of varieties are moderately resistant to CMV seed infection. While these varieties are still susceptible to CMV by aphid transmission, the build-up of inoculum in seed stock will be slower and the risk on CMV induced losses during virus favourable seasons will be lower.

References

There is a wealth of information on pulse viruses published on the internet by the agricultural departments of Western Australia (https://www.agric.wa.gov.au/), Queensland (https://www.daf.qld.gov.au/), Victoria (https://agriculture.vic.gov.au/) and New South Wales (https://www.dpi.nsw.gov.au/).

For more detailed information:

Makkouk KM, Kumari SG, van Leur JAG, Jones RAC (2014). Control of plant virus diseases in cool-season grain legume crops. Advances in Virus Research 90: 207-253.

Acknowledgements

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 authors would like to thank them for their continued support.

We would also like to acknowledge co-investment in this project by GRDC and NSW DPI through the Grains Agronomy Pathology Partnership (GAPP) and the outstanding collaboration and useful discussions with growers and agronomists during surveys.

Contact details

Joop van Leur
NSW Department of Primary Industries
4 Marsden Park Rd, Tamworth, 2340, NSW
Ph: 0427 928 018
Email: joop.vanleur@dpi.nsw.gov.au

Zorica Duric
NSW Department of Primary Industries
4 Marsden Park Rd, Tamworth, 2340, NSW
Ph: 02 6763 1154
Email: zorica.duric@dpi.nsw.gov.au

⍦ Varieties displaying this symbol beside them are protected under the Plant Breeders Rights Act 1994.

The economics of managing Ascochyta in chickpea when disease occurs at different growth stages and implications for spray timing

Leigh Jenkins[1], Kevin Moore[2] and Steven Harden[2]

[1] *NSW DPI, Trangie*
[2] *NSW DPI, Tamworth*

Key words

chickpea, Ascochyta, management, gross margin, profitability

GRDC codes

GAPP, Grains Agronomy & Pathology Partnership - A strategic agreement between GRDC and NSW DPI (DAN00213): BLG209 Pulse Integrated Disease Management – Northern NSW/QLD

Take home messages

- Impact of Ascochyta at different growth stages was investigated in a 2020 field experiment at Trangie with varieties Kyabra⫚, PBA HatTrick⫚ and PBA Seamer⫚
- Ascochyta caused yield losses from 100% to nil depending on when the disease occurred, and the variety grown
- Highest gross margin (GM)'s (> $800/ha) occurred with the lowest incidence of Ascochyta and also with the least susceptible variety, i.e. PBA Seamer⫚
- Manage Ascochyta early and grow varieties with best resistance to minimise impact of Ascochyta
- Follow current Ascochyta advice to maximise enduring profitability.

Why did we do this research?

Current management of chickpea Ascochyta in north central/northern NSW and southern QLD is based on results of field trials conducted mostly at Tamworth, grower experiences and feedback from agronomists. The Tamworth experiments have tried to simulate what happens in most farmers crops, i.e. initial infection occurs during the first post emergence rainfall event – accordingly, all the Tamworth experiments have established Ascochyta during that event. But what is the impact if infection occurs at later stages of growth and how does that affect management? The Trangie Agricultural Research Centre provided an opportunity to address that question – the soil and climate are typical of the Macquarie valley, a major chickpea production region. Overhead irrigation (lateral) and inoculation with conidia and infected spreader plants optimised infection and disease development.

Experiment details

Treatments

Aim: to assess the impact of Ascochyta blight (AB) disease occurring at three different growth stages, on yield of three chickpea varieties with different levels of AB resistance.

Ascochyta treatments (5):

1. **LOW (NIL)**: un-inoculated (NIL disease = CONTROL) plus foliar chlorothalonil fungicide (1.0 L/ha, chlorothalonil 720g/L) applied before rain or irrigation events

2. **HIGH**: inoculated with disease twice (at seedling (SDG, 3-4 nodes) and vegetative (VEG, 7-8 nodes) growth stages); NIL fungicide applied
3. **SDG**: inoculate with disease at seedling stage (3-4 nodes), allow disease to progress for 2-3 rain events to 7-8 nodes, then control disease for rest of season with chlorothalonil
4. **VEG**: protect plants from emergence to vegetative stage (7-8 nodes) with chlorothalonil; inoculate with disease and allow to progress for 2-3 rain events to first pods, then control disease with chlorothalonil
5. **POD**: protect plants from emergence to reproductive stage (first pods) with chlorothalonil, inoculate with disease and allow disease to progress through to harvest.

Variety treatments (3, and level of AB resistance):

1.	**Kyabra**⟇	VS = Very susceptible
2.	**PBA HatTrick**⟇	MS = Moderately susceptible
3.	**PBA Seamer**⟇	MR = Moderately resistant

Replication: 4 reps

Method

The experiment was conducted at Trangie Agricultural Research Centre in central west NSW, on a grey vertosol soil with access to overhead (lateral) irrigation. The experiment was sown as a randomised block design using a small plot seeder, with each plot 2m x 10m. Buffer plots were sown (with PBA Seamer⟇) at the same plot size between each treatment plot, to reduce the impact of inter-plot interference from Ascochyta inoculation and fungicide application. A back-pack sprayer with a 2m wide hand-held wand and 015 110 degree flat fan nozzles @ 50cm, was used to apply both Ascochyta inoculum and fungicide. Buffer plots received the full set of six fungicide applications.

Ascochyta disease was generated in treatment plots by a combination of two inoculation methods:
1. Ascochyta applied to whole plot as conidial suspension (600,000 conidia/mL), and
2. Ascochyta infected spreader plants transplanted to centre of plot.

Ascochyta treatments for each growth stage were applied just prior to either a forecast rain event, or irrigation.

Ascochyta treatment and fungicide were applied as follows:

up to 1 July	NIL fungicides applied prior to first disease treatment
1 July	**inoculation 1 - SDG & HGH treatments (pre-irrigation)**
9 July	fungicide 1 pre-rain - applied to LOW, VEG & POD (not SDG or HGH)
5 August	**inoculation 2 - VEG & HGH treatments (pre-rain)**
13 August	fungicide 2 pre-rain – applied to LOW, SDG & POD (not VEG or HGH)
9 September	fungicide 3 pre-rain – applied to LOW, SDG & POD (not VEG or HGH)
19 September	**inoculation 3 - POD treatment (pre-rain)**
29 September	fungicide 4 pre-rain – applied to LOW, SDG & VEG plots (not POD or HGH)
7 October	fungicide 5 pre-rain – applied to LOW, SDG & VEG plots (not POD or HGH)
21 October	fungicide 6 pre-rain – applied to LOW, SDG & VEG plots (not POD or HGH)

Site details & agronomy management

Sowing date: 26 May 2020
Harvest date: 27 November 2020
Seed treatment:

PBA Seamer⍦ pre-treated with thiram at purchase
Kyabra⍦ & PBA HatTrick⍦ treated with P-Pickel T® pre-sowing

Fertiliser at sowing: Granulock® Z @ 80 kg/ha

Inoculant: Group N, liquid inject at sowing

Target plant density: 30 plants/m^2

Actual establishment achieved:

Kyabra⍦ & PBA Seamer⍦ = 31 plants/m^2 (new seed)
PBA HatTrick⍦ = 21 plants/m^2 (retained seed ex 2019 harvest, poor storage conditions)

Buffers between treatment plots: PBA Seamer⍦ at 30 plants/m^2

Herbicide management:

pre-sow:	TriflurX® (trifluralin 480 g/L) @ 1.7 L/ha
PSPE:	Terbyne® Xtreme® (terbuthylazine 875 g/kg) @ 0.86 kg/ha
in-crop:	haloxyfop 520 @ 100 mL/ha + clethodim 240 @ 250 mL/ha

Fungicide application:

up to 6 chlorothalonil 720g/L @ 1.0 L/ha for LOW (NIL disease) treatment; total number of applications varied for each growth stage treatment

Insecticide & mouse management:

17 Sept	Affirm® (emamectin) insecticide @ 300 mL/ha (by plane)
6 Oct	Altacor® (chlorantraniliprole) insecticide @ 70 g/ha (by plane)
12 Oct	Zinc phosphide mouse bait @ 1.0 kg/ha (by plane)

Harvest management:

desiccation not required due to heatwave conditions from 15 November on
experiment harvested at 9% moisture content.

Pre-sowing rainfall (1/01/20 to 25/05/20): 351 mm

In-crop rainfall (26/05/20 to 27/11/20): 201 mm in 49 events

Plus in-crop irrigations: 20 mm (as 2 x 10 mm events)

Table 1. Summary of dates of disease inoculations and fungicide applications for 2020 Trangie chickpea Ascochyta management trial, relative to dates of subsequent rain or irrigation events.

2020 date	Disease inoculation	Fungicide application	Rain > 2.0mm	Irrigation (mm)	Cumulative >2.0mm # Events	Cumulative >2.0mm (mm)
01 Jul	SDG & HIGH		0.0	10.0	1	10.0
09 Jul		LOW, VEG & POD				
10-13 Jul			31.6		2	41.6
25-27 Jul			35.8		3	77.4
05 Aug	VEG & HIGH					
06-12 Aug			19.2		4	96.6
13 Aug		LOW, SDG & POD				
14-22 Aug			18.8		5	115.4
09 Sep		LOW, SDG & POD	3.8		6	119.2
14 Sep			0.0	10.0	7	129.2
19 Sep	POD					
19-25 Sep			33.8		8	163.0
29 Sep		LOW, SDG & VEG				
7 Oct		LOW, SDG & VEG	6.8		9	169.8
17 Oct			3.4		10	173.2
21 Oct		LOW, SDG & VEG				
23-25 Oct			17.6		11	190.8
4-5 Nov			7.8		12	198.6

Table 2. Log of operations for 2020 Trangie experiment on managing chickpea Ascochyta when disease occurs at different growth stages: Inoc = inoculation, DAS = days after sowing, DAI = days after inoculation.

	Ascochyta treatment			
Operation	**HIGH**	**SDG**	**VEG**	**POD**
Sow date	26/05/2020	26/05/2020	26/05/2020	26/05/2020
1st Inoc date	1/07/2020	1/07/2020	5/08/2020	19/09/2020
2nd Inoc date	5/08/2020	not done	not done	not done
1st Inoc DAS	36	36	71	116
2nd Inoc DAS	71	not done	not done	not done
Score 1 date	26/08/2020	26/08/2020	26/08/2020	26/08/2020
Score 2 date	1/10/2020	1/10/2020	1/10/2020	1/10/2020
Score 3 date	14/10/2020	14/10/2020	14/10/2020	14/10/2020
Score 1 DAI	56	56	21	na
Score 2 DAI	92	92	57	12
Score 3 DAI	105	105	70	25

Results

The effects of Ascochyta treatment, Variety and Ascochyta x Variety were highly significant ($P<0.001$) for all variables measured.

Disease incidence

The seedling, (SDG) and HIGH Ascochyta treatments were inoculated on 1 Jul 20, 36 days after sowing, DAS (Table 2). The HIGH treatment was inoculated a second time on 5 Aug (71 DAS) to maximise disease development (Table 2). This resulted in severe early disease (assessed on 26 Aug) in Kyabra⦹, moderate disease in PBA HatTrick⦹ and demonstrated the improved Ascochyta resistance in PBA Seamer⦹ (Table 3). The vegetative, VEG treatment was inoculated on 5 Aug (71 DAS), prior to which plants had been protected with foliar fungicide applied on 9 Jul. This regime produced low disease in Kyabra⦹and PBA HatTrick⦹ at the 1st and 2nd assessments (26 Aug, 1 Oct) and nil in PBA Seamer⦹ (Table 3). The podding, POD treatment was inoculated on 19 Sep (116 DAS), prior to which plants had been protected with foliar fungicides applied on 9 Jul, 13 Aug and 9 Sep. This regime produced nil disease in all three varieties at the 1st assessment (26 Aug) and low disease at the two subsequent assessments (1 Oct, 14 Oct) (Table 3). We assign the low incidence of Ascochyta with the POD treatment as a consequence of the three fungicide sprays applied before inoculation and less favourable seasonal conditions.

Table 3. Incidence of Ascochyta (% plot with disease) in Kyabra⦹, PBA HatTrick⦹ and PBA Seamer⦹ chickpeas when disease was established at different growth stages but controlled before and after; l.s.d. (P=0.05) for Scores 1, 2 & 3 = 13.26%, 8.63% & 11.74%, respectively.

Variety	Ascochyta treatment	Score 1 26 Aug 20	Score 2 01 Oct 20	Score 3 14 Oct 20
Kyabra ⦹	LOW	1.3	0.0	5.2
PBA HatTrick ⦹	LOW	0.0	0.0	11.2
PBA Seamer ⦹	LOW	0.0	0.0	3.0
Kyabra ⦹	SDG	92.0	57.5	70.0
PBA HatTrick ⦹	SDG	37.5	22.5	40.0
PBA Seamer⦹	SDG	0.0	0.0	2.8
Kyabra ⦹	VEG	8.8	12.5	31.2
PBA HatTrick ⦹	VEG	5.0	8.8	22.5
PBA Seamer⦹	VEG	0.0	0.0	3.0
Kyabra ⦹	POD	0.0	0.5	7.8
PBA HatTrick ⦹	POD	0.0	2.5	5.2
PBA Seamer⦹	POD	0.0	0.0	3.8
Kyabra⦹	HIGH	88.8	100.0	100.0
PBA HatTrick ⦹	HIGH	32.5	52.5	57.5
PBA Seamer ⦹	HIGH	0.0	0.0	4.0

This approach, i.e. inoculating Ascochyta at different times and protecting before and after, allowed us to determine the impact of disease at those stages on yield.

Impact on yield

Table 4. Effect of Ascochyta on grain yield, gross margin (GM) and yield loss for three chickpea varieties when disease occurs at different growth stages; l.s.d. (*P*=0.05) yield 286 kg/ha. GM is based on chickpea price of $600/t, fungicide product $16/ha/application, fungicide ground rig application $5/ha, other production costs $300/ha.

Variety	Ascochyta treatment	Yield (kg/ha)	GM ($/ha)	% Yield loss (kg/ha)
Kyabraⓟ	LOW	1878	701	0
PBA HatTrickⓟ	LOW	1840	678	0
PBA Seamerⓟ	LOW	2138	857	0
Kyabraⓟ	SDG	10	-399	99
PBA HatTrickⓟ	SDG	965	174	48
PBA Seamerⓟ	SDG	2080	843	3
Kyabraⓟ	VEG	1483	506	21
PBA HatTrickⓟ	VEG	1504	518	18
PBA Seamerⓟ	VEG	2211	943	-3
Kyabraⓟ	POD	2041	862	-9
PBA HatTrickⓟ	POD	1880	765	-2
PBA Seamerⓟ	POD	2101	898	2
Kyabraⓟ	HIGH	0	-300	100
PBA HatTrickⓟ	HIGH	234	-160	87
PBA Seamerⓟ	HIGH	1903	842	11

Grain yield (Table 4) ranged from nil (Kyabraⓟ HIGH) to over 2 t/ha (all PBA Seamerⓟ treatments). For the very susceptible Kyabraⓟ, lowest yields occurred with the SDG and HIGH treatments with yield losses of 99% and 100% respectively. The moderately susceptible PBA HatTrickⓟ also had lowest yields for SDG and HIGH treatments, with losses of 48% and 87% respectively (Table 4). The least susceptible variety PBA Seamerⓟ only lost 3% and 11% yield from SDG and HIGH treatments.

Gross Margins, GM

The highest GM's occurred with the lowest incidence of Ascochyta (LOW) and also with the least susceptible variety PBA Seamerⓟ (Table 4). All PBA Seamerⓟ treatments, including the one with most Ascochyta, (HIGH) had GM over $800/ha (Table 4). However, the experiment showed that controlling Ascochyta in the very susceptible Kyabraⓟ is profitable with a GM of $701/ha and $862/ha for the LOW and POD treatments respectively.

Conclusions

Generating chickpea Ascochyta at different stages of growth showed when and how disease affects yield. The impact of disease on a chickpea crop depends primarily on when the disease occurs, how it is managed, and the variety grown. Allowing Ascochyta to establish early in the life of your crop results in greatest impact on yield and lowest profitability, even if the disease is subsequently controlled with foliar fungicides. This is especially true for very susceptible and moderately susceptible varieties. Your best option for minimising impact of Ascochyta on chickpea production and maximising profitability is to follow current recommendations (Moore and Heuston, 2020) by controlling disease early and growing the least susceptible variety. This approach will also reduce the build-up and carryover of Ascochyta inoculum for your and your neighbour's future chickpea crops.

References

Kevin Moore and Penny Heuston (2020) Managing Ascochyta blight in chickpeas in 2020. NSW DPI Fact Sheet, May 2020
https://www.dpi.nsw.gov.au/__data/assets/pdf_file/0015/1220271/managing-ascochyta-blight-in-chickpeas-in-2020.pdf

Acknowledgements

This research is a component of the "Pulse Integrated Disease Management – Northern NSW/QLD" project (BLG209), as part of the Grains Agronomy & Pathology Partnership (GAPP) between GRDC and NSW DPI. The research undertaken in this project is made possible by the significant contributions of growers through both trial cooperation and the support of the GRDC. We would like to thank GRDC, growers and their agronomists for their continued support of this research.

Leigh Jenkins and Kevin Moore would also like to acknowledge and thank NSW DPI Technical support staff for their assistance with laboratory preparation and field work, including Scott Richards, Liz Jenkins and Joanna Wallace at Trangie ARC; and Paul Nash and Gail Chiplin at Tamworth AI.

Contact details

Leigh Jenkins & Kevin Moore
NSW DPI, Trangie Agricultural Research Centre & Tamworth Agricultural Institute
Ph: LJ 0419 277 480, KM 0488 251 866
Email: leigh.jenkins@dpi.nsw.gov.au, kevin.moore@dpi.nsw.gov.au

⯑ Varieties displaying this symbol beside them are protected under the Plant Breeders Rights Act 1994.

® Registered trademark

CBA Captain⑴: a new desi variety for the northern region

Kristy Hobson[1], Nicole Dron[1], Merrill Ryan[2], William Martin[2], Neroli Graham[1], Annie Warren[1], Jenny Davidson[3], Sean Bithell[1], Kevin Moore[1], Jenny Wood[1], Glenn Lendon[1] and Georgina Pengilley[1]

[1] NSW DPI Tamworth
[2] DAF Warwick
[3] SARDI Adelaide

Key words

chickpea variety, desi, yield, disease resistance, Ascochyta blight, Phytophthora root rot, phenology, grain quality

GRDC codes

DAN00094, DAN00151, DAN00176, DAN00212, BLG205, BLG209, BLG111, 9177999

Take home message

CBA Captain⑴ is a new broadly adapted desi chickpea variety for the northern region and other chickpea growing areas of Australia. The variety was evaluated as CICA1521 and has higher yields in northern NSW and southern QLD than PBA HatTrick⑴. CBA Captain⑴ has a medium seed size and is expected to have similar disease ratings to PBA HatTrick⑴. Seed of CBA Captain⑴ is available in the northern region for the 2021 season from the following seed partners; Galleon Grains, PB Agrifood, PB Seeds and Woods Seeds.

Significant yield advantage over PBA HatTrick⑴

CBA Captain⑴ has been included in National Variety Trials (NVT) since 2015. During this six-year period there have been two favourable seasons in north west NSW (2016 and 2020) and a number of dry seasons (2018 and 2019). CBA Captain⑴ has shown great consistency in yield despite the highly variable seasons. In the southern areas of north-west NSW, CBA Captain⑴ has consistent yield gains over PBA HatTrick⑴ (Table 1). At the time of writing this paper, the long term yield reporter using 2020 trials were not available. Readers are encouraged to visit nvtonline.com.au.

Table 1. Long term yield (2015-2019) of CBA Captain⊕ and current chickpea varieties, expressed as a % of the mean yield, in NVT in Trangie, Narromine and Coonamble. The mean yield of all varieties in each of the 5 contributing trials was used to assign that trial to a 'yield grouping'. This enables varietal performance to be better evaluated in different yield situations

		Yield group (t/ha)			
		1	**1.5**	**2.0**	**3.0**
	Mean yield (t/ha)	0.91	1.25	1.84	2.96
Variety	No trials in total & for each yield group	2	1	1	1
CBA Captain⊕	**5**	**108**	**110**	**110**	**112**
Kyabra⊕	5	113	98	97	68
PBA Boundary⊕	5	110	101	103	89
PBA Drummond⊕	1	107			
PBA HatTrick⊕	5	101	95	96	88
PBA Seamer⊕	5	95	102	102	115

Source: https://app.nvtonline.com.au/lty/table/chickpea-desi/nsw/nw/trangie,narromine,coonamble/?lty-type=yield&stacked=1

Table 2. Long term yield (2015-2019) of CBA Captain⊕ and current chickpea varieties, expressed as a % of the mean yield, in NVT at Bellata, north west NSW. The mean yield of all varieties in each of the 4 contributing trials was used to assign that trial to a 'yield grouping'. This enables varietal performance to be better evaluated in different yield situations

		Yield group (t/ha)		
		1.0	**1.5**	**2.0**
	Mean yield (t/ha)	0.96	1.18	1.65
Variety	No trials in total & for each yield group	2	1	1
CBA Captain⊕	**4**	**109**	**121**	**104**
Kyabra⊕	4	113	68	96
PBA Boundary⊕	4	104	95	96
PBA Drummond⊕	1	115		
PBA HatTrick⊕	4	97	86	96
PBA Seamer⊕	4	96	116	102

Source: https://app.nvtonline.com.au/lty/table/chickpea-desi/nsw/ne/bellata/?lty-type=yield&stacked=1

Ascochyta blight

CBA Captain⦸ has undergone Ascochyta blight (AB) testing in the field at Tamworth and Horsham as well as single isolate testing under controlled conditions in Adelaide and Tamworth.

An increase in the aggressiveness of the AB pathogen has been observed both in the northern and southern regions (Ford et al., 2018). Increased levels of disease have been recorded on CBA Captain⦸ and other varieties such as PBA Seamer⦸ from these isolates collected in 2017 compared to isolates collected in 2015 (Table 3). The distribution of the more aggressive isolates in the northern region is currently unknown, due to the reduced chickpea area and dry seasons over the past two years, however a cautious disease rating from the NVT pulse disease rating system for CBA Captain⦸ is expected. For northern isolates this is likely to be a Moderately Susceptible rating for Ascochyta blight. At the time of writing this paper the 2021 NVT pulse disease ratings were unavailable. Readers are encouraged to visit nvtonline.com.au for updated AB ratings.

Table 3. Mean disease index of chickpea varieties in single isolate AB screening conducted at Adelaide. The index is calculated as the sum of (% main stems broken + % of stems with lesions + % side branches with disease + % leaves with disease) divided by 4. (0 = healthy plant, 100 = heavily diseased plant)

	Isolate collection location and year							
Name	Yallaroi NNSW 2015	Curyo VIC 2015	Graman NNSW 2016	Curyo VIC 2016	Gurley NNSW 2017	Pt Broughton SA 2017	Gurley NNSW 2017	Curyo VIC 2018
CBA Captain⦸	**40**	**50**	**77.1**	**76.3**	**29.2**	**26.7**	**67.9**	**89.6**
Kyabra⦸			100	100	100	100	100	100
PBA Boundary⦸	81.3	67.9	100	100	77.1	55.4	90	100
PBA Drummond⦸	97.1	100	100	100	66.7	97.5	83.3	100
PBA HatTrick⦸	87.5	61.7	86.3	94.2	66.7	48.8	67.9	68.3
PBA Seamer⦸	31.7	37.5	83.8	90.8	29.2	39.2	65.4	84.6
Lsd	*20.6*	*27.2*	*18.1*	*16.2*	*24.7*	*19.2*	*26.6*	*28.6*

Phytophthora root rot

CBA Captain⦸ was included in Phytophthora root rot (PRR) yield loss trials conducted at Warwick QLD, over several years (Table 4). Yield losses for CBA Captain⦸ from PRR in these trials have ranged from 38.7 to 93.4 %. Similar variability in yield loss has also been observed for PBA HatTrick⦸; an explanation of the seasonal impacts on yields and varietal PRR disease rankings is provided in Bithell et al., 2018. In 2020, NVT pulse disease rating testing was conducted for PRR for the first time. It is expected that a review of all variety ratings for PRR will be conducted to align with the NVT disease rating definitions. The 2020 ratings were not available at the time of writing this report, please see nvtonline.com.au for updated ratings.

Table 4. Yield (t/ha) in the absence of PRR and yield loss (%) from PRR across 2016 to 2018 for CBA Captain and other current chickpea varieties. Adapted from Bithell et al., 2018, Bithell et al., 2019

	2016		2017		2018	
Name	Yield (t/ha) in the absence of PRR	% yield loss from PRR	Yield (t/ha) in the absence of PRR	% yield loss from PRR	Yield (t/ha) in the absence of PRR	% yield loss from PRR
CBA Captain	**4.06**	**75.1**	**2.74**	**93.4**	**1.94**	**38.7**
PBA Boundary	3.98	95.2	2.63	82.5		
PBA Drummond					2.49	68.1
PBA HatTrick	4.02	90.0	3.31	78.2	2.28	40.5
PBA Seamer	4.08	76.7	3.23	90.4	2.81	61.5
Yorker	4.06	68.3	3.50	97.3	2.84	40.1

Phenology and other agronomic traits

CBA Captain is early flowering when sown in the mid-May to mid-June sowing window, approximately six days earlier than PBA HatTrick (Table 5). Flowering data collected from early May sown chilling tolerance trials (BLG111) indicates that CBA Captain can flower up to 24 days earlier than PBA HatTrick depending on winter daytime temperatures. An increased understanding of the drivers of chickpea phenology is expected with new GRDC investments in this area. Although there is some data indicating that CBA Captain may produce pods earlier in some environments (e.g. Kingaroy, 2019), it is expected that days to first pod is similar to current varieties.

Table 5. Phenology data (2017-2020) collected for CBA Captain and current chickpea varieties from breeding and chilling tolerance trials in northern NSW and southern QLD

		CBA Captain		Kyabra		PBA Drummond		PBA HatTrick		PBA Seamer	
Sowing date	Location	DTF	DTP	DTF	DTP	DTF	DTP	DTF	DTP	DTF	DTP
15/6/2017	Spring Ridge	**102**		103				103		102	
23/5/2018	Moree	**91**		96		97		98		97	
7/5/2018	Tamworth	**101**	**134**	112	134	120	134	125	135	126	134
12/6/2018	Tamworth	**96**	**108**	99	108	97	108	102	108	99	
13/5/2019	Narrabri	**77**	**108**	83	106			80	108		
15/5/2019	Breeza	**106**	**122**	105	123			105	122		
17/5/2019	Kingaroy	**79**	**91**	82	95			81	96		
10/06/2020	Narrabri	**86**		88		88		89		88	

DTF = days to flower from sowing
DTP = days to pod from sowing

CBA Captain has early to mid-maturity, earlier than PBA HatTrick. CBA Captain has an erect plant type with good height to lowest pod and plant height. Under the high biomass producing conditions of 2016 and 2020, CBA Captain had less lodging than PBA HatTrick at seven sites across northern NSW and southern QLD (Table 6).

Table 6. Mean lodging score at northern NSW and southern QLD breeding sites in 2016 and 2020 for CBA Captain and current chickpea varieties. 1 = erect, 9 = flat.

Location	Year	CBA Captain	Kyabra	PBA Drummond	PBA HatTrick	PBA Seamer
Edgeroi	2016	**3.7**	2.3		5.3	5.3
North Star	2016	**4.3**	3.0		7	3.3
Rowena	2016	**5.3**	5.3		6.7	6.3
Warwick	2016	**6.0**	6.3		6.3	5.3
Warra	2016	**3.3**	2.0		6.0	2.7
Roma	2016	**3.7**	3.0		6.0	4.7
Rowena	2020	**1.7**	1.3	1.3	3.0	1.0

In December 2020, Tamworth Agricultural Institute received more than 240 mm of rain. Plots of CBA Captain were observed to have remained standing with good harvestability compared to PBA HatTrick which had lodged considerably.

In 2019 and 2020, large seed multiplications and demonstration blocks of CBA Captain were harvested by commercial harvesters throughout the northern region. No negative feedback regarding the harvestability of CBA Captain were reported.

Grain quality

CBA Captain has a yellow-brown seed coat and angular seed shape, not unlike PBA HatTrick. The seed size of CBA Captain is larger than PBA HatTrick, similar to PBA Seamer and PBA Drummond but smaller than Kyabra (Table 7). CBA Captain has a higher or similar split yield than PBA HatTrick and PBA Drummond at six sites across southern QLD and northern NSW.

Table 7. Seed size (grams per 100 seeds) and split yield % (SY%) for CBA Captain and other current chickpea varieties at six sites in northern NSW and southern QLD

	CBA Captain		Kyabra		PBA Drummond		PBA HatTrick		PBA Seamer	
Site Year	100 SW	SY%	100 SW	SY%	100 SW	SY%	100 SW	SY%	100 SW	SY%
Roma 2017	**19.8**	**52.9**	21.9	72.5			20.1	40.8	20.8	44.8
Spring Ridge 2017	**22.5**	**53.2**	24.6	45.1			18.9	44.3	21.3	55.7
Warra 2017	**21.7**	**72.3**	24.2	67.5			22.5	70.5	24.1	73.7
Moree 2018	**21.5**	**41.2**	25.1	46.9	22.2	36.7	20.2	42.9	21.5	42.0
North Star 2018	**23.7**	**46.0**	27.2	64.1	24.0	39.7	22.0	39.2	23.5	45.3
Warra 2019	**22.3**	**50.7**	24.2	32.6	22.4	38.0	21.8	37.4	22.2	38.7

100SW = grams per 100 seeds
SY% = split yield % (yield of dhal using a standard SKE milling method without pre-conditioning seeds; Wood et al 2008).

Seed partners

CBA Captain will be delivered to the northern region through the following seed partners; Galleon Grains, PB Agrifood, PB Seeds and Woods Seeds to distribute seed to QLD and NSW.

Acknowledgements

CBA Captain was developed by the PBA Chickpea program (led by NSW Department of Primary Industries). The partners of the PBA Chickpea program were: GRDC, NSW DPI, Department of Agriculture and Fisheries (QLD), Agriculture Victoria and the South Australian Research and Development Institute.

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

References

Bithell S., Kelly L., Hobson K., Harden S., Martin W., Chiplin G. and Moore K. (2018). https://grdc.com.au/resources-and-publications/grdc-update-papers/tab-content/grdc-update-papers/2018/03/phytophthora-in-chickpea-varieties-2016-and-2017-trials-resistance-and-yield-loss

Bithell S., Hobson K., Martin W., Ryan M., Harden S. and Moore K. (2019). https://grdc.com.au/resources-and-publications/grdc-update-papers/tab-content/grdc-update-papers/2019/02/latest-research-on-chickpea-phytophthora-root-rot-reduced-yield-losses-in-crosses-with-wild-cicer-relatives

Ford R., Moore K., Sambasivan P., Mehmood Y., Hobson K., Walela C., Brand J. and Davidson J. (2018). https://grdc.com.au/resources-and-publications/grdc-update-papers/tab-content/grdc-update-papers/2018/03/why-adhering-to-integrated-ascochyta-rabiei-management-strategy-is-now-more-important-than-ever

Wood J.A., Knights E.J. and Harden S. (2008). Milling performance in desi-type chickpea (*Cicer arietinum* L.): effects of genotype, environment and seed size. Journal of the Science of Food and Agriculture, 88, 108-115. https://onlinelibrary.wiley.com/doi/full/10.1002/jsfa.3053

Contact details

Kristy Hobson
Chickpea Breeding Australia
New South Wales Department of Primary Industries
4 Marsden Park Rd
Calala NSW 2340
Ph: 02 6763 1174
Email: Kristy.hobson@dpi.nsw.gov.au

Varieties displaying this symbol beside them are protected under the Plant Breeders Rights Act 1994

How wide is the distribution of RWA in northern NSW and is sorghum an alternative summer host?

Zorica Duric, NSW DPI

Key words

Diuraphis noxia, presence, over-summer, host range, symptoms

GRDC codes

BLG204, BLG214

Take home messages

- Tamworth (NSW) is the most northern site where Russian wheat aphid (RWA) has been confirmed in Australia
- RWA has been found in barley, wheat, durum wheat and barley grass in northern NSW to date
- Millet (variety Jandowae) is not a suitable host plant for RWA
- The RWA survival in sorghum in northern NSW is likely to depend on climatic conditions. High temperature and humidity may supress RWA survival and reproduction
- Glasshouse trials showed the highest number of adults and nymphs in wheat (Lancer⦶) and barley (Commander⦶), followed by oats (Yiddah⦶, Mannus⦶, Nile), sorghum (Sentinel IG) and triticale (Endeavour⦶)
- The typical symptoms of RWA presence are found in its primary hosts (barley and wheat), while little or no symptoms were observed in its secondary hosts (oats, sorghum, triticale).

Background

Russian wheat aphid (RWA) - *Diuraphis noxia* (Kurdjumov) (Homoptera: Aphididae) is a worldwide known pest of cereals. It originates from central Asia, the Middle East and southern Russia and its presence has been confirmed across cereal growing regions in Asia, Europe, Africa, North and South America and, since 2016, in Australia (Kindler & Springer, 1989, Hughes, 1996, Yazdani et al., 2018). The RWA primary hosts are barley, wheat and durum wheat, but it can infect triticale, rye and oats. Its host range also includes winter, and some summer wild grasses.

The RWA is easily distinguishable from other cereal aphids such as the oat aphid (*Rhopalosiphum padi*), corn aphid (*Rhopalosiphum maidis*), and rose-grain aphid (*Metopolophium dirhodum*). RWA is a pale green, spindle-shaped small aphid which is often covered with fine wax. It has dark eyes, antennae shorter than half its body length, almost invisible cornicles and an appendage above the cauda which gives it the appearance of having two tails.

It feeds on plant sap and affects host plants by injecting toxins while probing and feeding. In response, cereal plants start to develop various symptoms including chlorosis, necrosis, wilting, stunting, leaf streaking with whitish, yellow and purple longitudinal leaf markings, and rolled leaves. If probing occurs during the heading growth stage, trapped awns or bleached heads could develop or flowering may not occur.

Wheat and barley can provide a suitable habitat for RWA for a large part of the year. However, wild grasses are also very important for persistence of RWA populations over summer and providing a bridge for the infestation of autumn cereals.

This study was conducted to explore the distribution of RWA in northern NSW and possibility of migration, survival and reproduction on sorghum, the major summer cereal grown in the northern region.

Methods

The distribution survey was carried out on autumn sown cereals, volunteer crops and winter grasses in order to determine the presence of RWA in northern NSW. Collected samples were: wheat (*Triticum aestivum*), barley (*Hordeum vulgare*), durum wheat (*Triticum durum*), barley grass (*Hordeum leporinum*), prairie grass (*Bromus cathartions*), wild oats (*Avena sativa*), couch grass (*Cynodon dactylon*), Johnson grass (*Sorghum halepense*), phalaris (*Phalaris aquatica*), liverseed grass (*Urochloa panicoides*), and Queensland blue grass (*Dichanthium sericeum*). Samples were cut low to ground level, collected and transported in plastic bags, and placed in Berlese funnels in the laboratory for 3-4 days. Extracted insects were observed under stereomicroscope and the results were included in RWA interactive map (http://www.cesaraustralia.com/sustainable-agriculture/rwa-portal/).

The preliminary RWA field study was conducted on irrigated leased land near the Tamworth Agricultural Institute (TAI), to examine over-summering and reproduction of RWA on the major summer grains in northern NSW: sorghum (Sentinel IG) and millet (var. Jandowae). The experiment was set up in summer 2019/2020 under aphid proof tents, in two repetitions. The first inoculation of RWA on both sorghum and millet was at the 3-leaf growth stage. One plant per tent was inoculated with 10 wingless aphids. The second inoculation was at 5-leaf growth stage.

The 2020 RWA winter grains study was established to analyse the possible migration of aphids from winter to summer cereal (sorghum). Aphid-proof tents were half sown with wheat (LancerΦ) or barley (CommanderΦ). The other half of each tent was sown with sorghum (Sentinel IG) at the end of the 2020 winter season, before the wheat and barley were ready to be harvested. At the tillering growth stage 10 wheat/barley plants in each tent were infested with 10 wingless RWA. The RWA colonisation and distribution on winter, as well as migration onto summer grain hosts were studied till December 2020.

Damage analysis and host preference of RWA was investigated in the TAI glasshouse on barley (CommanderΦ), wheat (LancerΦ), oats (YiddahΦ, MannusΦ, Nile), sorghum (Sentinel IG), and triticale (EndeavourΦ). Ten plants of each host were infested with 10 wingless adults. Adults and nymphs counts were observed 2, 7 and 14 days after infestation.

Results and discussion

Distribution survey

Since the first report of RWA in Australia in 2016 (Yazdani et al., 2018), it has spread rapidly through the eastern grain belt. The data from 2019 showed that Tamworth was the most northern site where RWA has been confirmed in Australia (Figure 1). Out of 21 collected samples in 2019 there were 3 positives in barley, 1 positive in wheat, 1 positive in durum wheat, and 3 positives in barley grass. In a further survey in 2020, 58 cereal and grass samples collected from various sites in Liverpool Plains showed no positives. The RWA population suppression could be explained by the hot dry 2019 summer and the lack of over-summer green bridge. 2019 was Australia's warmest and driest year on record and the dry summer was followed by the coolest and wettest autumn in NSW since 2012 (BOM, 2020 a, b).

A temperature over 20°C is unfavourable for RWA and it cannot survive at temperatures of over 37°C. Furthermore, RWA prefers drier climates, where summer rainfall is 300-400 mm. Heavy rainfall can wash aphids off the upper leaves and 30mm rainfall may cause 50% mortality (Hughes, 1996, GRDC, 2017).

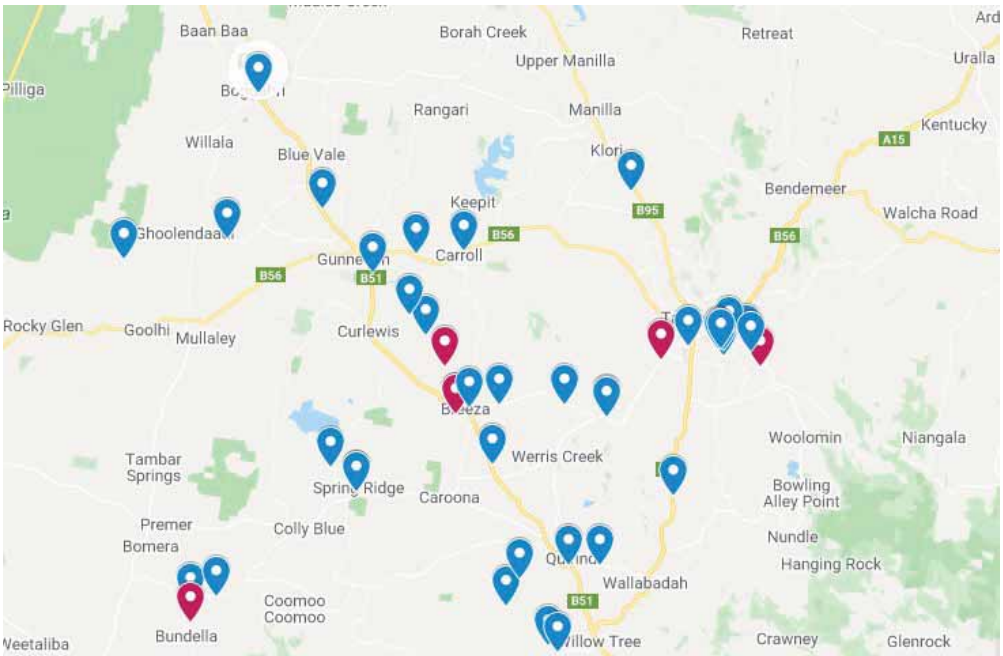


Figure 1. Detail of map of RWA positive (●) and negative (●) samples in 2019 and 2020

RWA sorghum field study

The second inoculation in the preliminary RWA field study in December 2019 was successful for sorghum, but not for millet. The RWA did not manage to establish on millet, and therefore the millet (var. Jandowae) was not found as suitable host plant for RWA.

The aphids were initially observed at the inoculation points of the upper side of the sorghum flag leaf, before migrating to the back side of the leaf, mainly next to the central leaf vein (Figure 2) and starting reproduction. During January 2020, aphids migrated to neighbouring leaves in the lower canopy, likely due to the better protection from high temperatures and a population up to 30 individuals developed on the inoculated tiller. When the sorghum reached the flowering/grain filling stage (end of February 2020), established RWA colonies were observed on 3 neighbouring plants out of the 10 plants inside the tents. Although no symptoms were observed, this result demonstrates that RWA was able to survive on sorghum plants during the 2019/20 summer. Similarly, Harvey and Kofoid (1993) found three sorghum lines which supported RWA for at least a month. Since RWA is typically considered a minor pest of sorghum, there is a possibility that the aphids would have moved to more favourable host plants in an open field study.

Figure 2. RWA colony on the back side of sorghum leaf

RWA winter grains field study

An established population of 3-10 adults of RWA was noticed on all infested plants six days after inoculation on 12 June 2020. The first symptoms were observed in 1-3 out of 10 infested barley/wheat plants per tent. The plant response to the RWA toxic secretion was fast and only a few aphids can result in symptoms in less than 7 days. The symptoms were characteristic white to purple stripes, and leaf-rolling on both wheat and barley leaves. At 15 days after infestation, both wingless and winged forms were found in colonies and 5-7 additional plants were infested. One month after infestation 30% of plants showed typical symptoms and 2 months after infestation (6 August 2020) all plants inside the tents were infested with moderate to high populations. During August and September typical symptoms developed in both barley and wheat including rolled leaves, stunted growth, trapped awns, and twisted and distorted heads.

By mid-September 2020, both barley and wheat became unfavourable hosts due to ripening and, since the majority of RWA dispersal occurs by flying (Hughes, 1996), high numbers of winged aphids were noticed inside the tents. After the sorghum emerged on 13 October, the winged aphids migrated from both barley and wheat to the young sorghum plants and started to reproduce. Both winged and wingless aphids and their progeny were observed on the young sorghum plants at the same time (Figure 3a). Symptoms such as red tips on the leaves and patches at the place of feeding (Figure 3b) were observed. Similar to that reported by Harvey and Kofoid (1993), the aphids successfully infested and damaged the susceptible sorghum plants. The aphids first infested the leaf tips and edges, and then moved to the leaf base leaving skins behind. This type of damage could cause plant death in a short time, however, in October 2020 the daily temperatures inside the tent were too hot for RWA development. Temperatures above 40°C were recorded for 2-6 hours in both the morning and early afternoon for most of October, with the highest, 53.15°C, recorded on 21 October. The RWA population on sorghum quickly decreased, as they cannot survive long enough to reproduce at high temperatures. A few surviving individual adults with a small number progeny were observed on sorghum in a couple of tents at the end of October. No aphids were observed in November and the sorghum plants recovered. However, this result indicates that with favourable climatic conditions, RWA could use sorghum as alternate host in late spring, early summer.

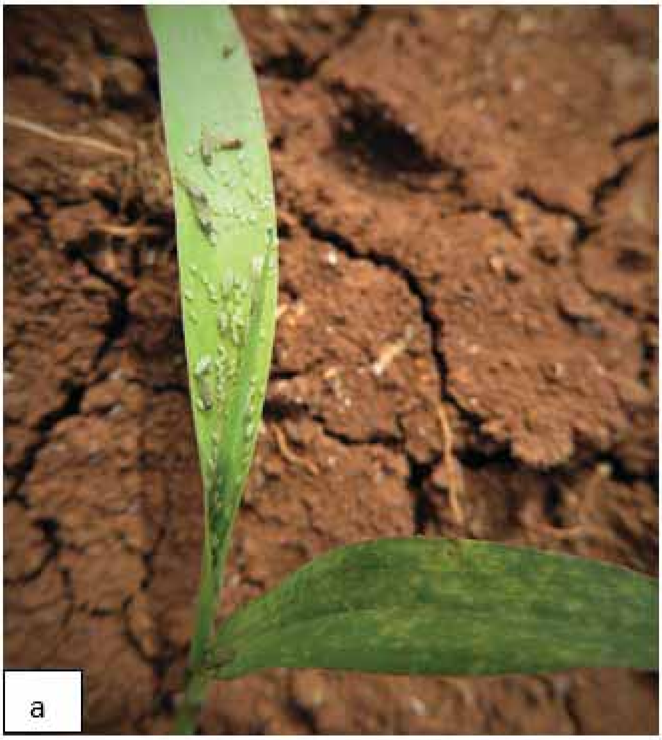


Figure 3. RWA infestation (a) and damage (b) of young sorghum plant

RWA host preference study

In the host preference study, at 14 days after infestation (DAI), the RWA population was best established on wheat, with a mean number of 49 adults and 148 nymphs, and barley (42 adults and 140 nymphs). The highest mean number of established aphids in oats were found on variety Yiddah⦶ (9 adults and 28 nymphs), then Mannus⦶ (9 adults and 26 nymphs) and Nile (5 adults and 16 nymphs). The lowest mean numbers were in sorghum (9 adults and 9 nymphs) and triticale (4 adults and 9 nymphs) (Figure 4). Sorghum appeared as one of the less favourable host plants. However, as the aphid did not establish on all the tested sorghum plants, further studies are required in order to achieve a clearer result.

Symptoms occurred 7 DAI on wheat and barley, but not until later on other host plants. At 14 DAI, both barley and wheat leaves were covered with aphids and their skins, with the symptoms including yellow stripes and leaf rolling. Also, due to the crowding, many winged adults were noticed. The less preferred hosts (oats, sorghum, triticale) showed little or no symptoms. The oat leaves had hardly visible, chlorotic yellow lines with the aphids concentrated on leaf tips, on the upper and back side of the leaf. On triticale, the RWA population remained at the base of the young leaves and inside rolled leaves. Yellow stripes were hardly visible. On sorghum, RWA were usually based on the upper side of the leaf. Red patches developed at the place of feeding on the plants where the RWA became established.

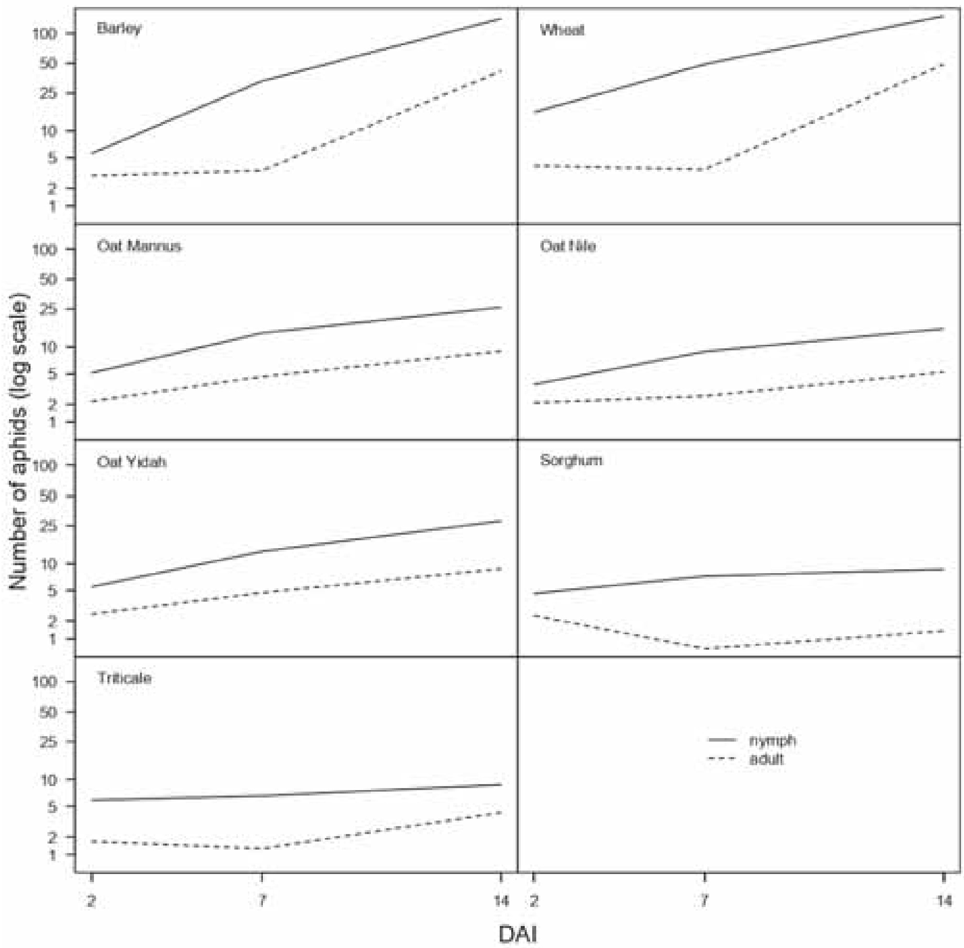


Figure 4. Mean numbers of adults and nymphs of Russian wheat aphid (RWA) on different hosts 2, 7 and 14 days after infestation (DAI)

Wheat and barley were found to be the most preferable host for RWA. Compared with wheat and barley, the other studied plants were poor hosts. However, the host range of RWA includes more than 140 species (GRDC, 2017), and there is a great possibility for RWA to maintain its population using these secondary hosts, including oat, triticale and sorghum, during the off-season.

Conclusion

RWA has been found in barley, wheat, durum wheat and barley grass in the Tamworth region in 2019.

Sorghum is the major summer cereal grown in the northern region with an average area sowed of 160,000 ha in northern NSW and 470,000 ha in Queensland. The results from this study show that sorghum can provide a host for RWA, especially on young plants in late spring and at the beginning of summer. However, additional field studies are needed to address its risk to the northern wheat industry.

While RWA forms well-established colonies and typical symptoms on its primary hosts (barley and wheat), the aphid can also maintain small colonies and develop symptoms that include red patches at the place of feeding on sorghum leaves, and hardly visible yellow lines and rolled leaves on oat and triticale.

References

BOM (2020a). Annual climate statement 2019, Australian Government, Bureau of Meteorology, http://www.bom.gov.au/climate/current/annual/aus/ [Issued 9 January 2020]

BOM (2020b). New South Wales in autumn 2020: wet and cool, Australian Government, Bureau of Meteorology, http://www.bom.gov.au/climate/current/season/nsw/archive/202005.summary.shtml#:~:text=Autumn%202020%20in%20New%20South,the%20north%20and%20south%20coasts [Issued 1 June 2020]

GRDC (2017). Tips and tactics Russian wheat aphid, Grow Notes, Southern Region, https://grdc.com.au/__data/assets/pdf_file/0025/289321/GRDC-Tips-and-Tactics-Russian-Wheat-Aphid.pdf

Harvey TL and Kofoid KD (1993). Reproduction and Survival of the Russian Wheat Aphid (Homoptera: Aphididae) on Sorghum, Journal of the Kansas Entomological Society, 66, 1, 81-85.

Hughes RD (1996). A synopsis of information on the Russian wheat aphid, *Diuraphis noxia* (Mordwilko), Revised Edition, Division of Entomology, Technical Paper No. 34, CSIRO Australia, ISBN No. 0643048723.

Kindler SD and Springer TL (1989). Alternate Hosts of Russian Wheat Aphid (Homoptera: Aphididae), Journal of Economic Entomology, 82, 5, 1358–1362, https://doi.org/10.1093/jee/82.5.1358.

Yazdani M, Baker G, DeGraaf H, Henry K, Hill K, Kimber B, Malipatil M, Perry K, Valenzuela I and Nash MA (2018). First detection of Russian wheat aphid *Diuraphis noxia* Kurdjumov (Hemiptera: Aphididae) in Australia: a major threat to cereal production, Austral Entomology, 57, 410–417.

Acknowledgements

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.

Contact details

Zorica Duric
Department of Regional NSW
4 Marsden Park Rd, Tamworth, 2340, NSW
Ph: 02 6763 1154
Email: zorica.duric@dpi.nsw.gov.au

⦹ Varieties displaying this symbol beside them are protected under the Plant Breeders Rights Act 1994.

How heat tolerant are our current wheat varieties?

Rebecca Thistlethwaite[1], Anowarul Bokshi[1], Sang He[2], Reem Joukhadar[2], Daniel Tan[1] and Hans Daetwyler[2], Richard Trethowan[1]

[1] The Plant Breeding Institute, The University of Sydney
[2] Agriculture Victoria, AgriBio, Bundoora

Key words

wheat, heat tolerance, genomic selection, phenotyping, pre-breeding, current varieties

GRDC code

US00081

Take home message

Some Australian cultivars are significantly more heat tolerant than others when heat stress is applied at anthesis and grain filling growth stages. Materials identified and developed through this project provide new genetics for heat tolerance that can be used to improve the heat tolerance of current varieties.

Aims

This research compared the heat tolerance of current commercial wheat varieties under elevated day and night temperatures during anthesis (Zadok 60) and grain fill (Zadok 73) growth stages.

Introduction

Extreme heat events may become 85% more frequent and 57% longer in the coming decades due to the increasing impact of climate change (Syktus et al; 2020). Wheat is highly sensitive to extreme heat during anthesis (flowering) and grain fill growth stages. This can have a significant impact on final crop yield and quality. Heat stress at anthesis often affects pollen fertility which reduces the number of kernels produced by each spike. For every one degree rise in the average maximum temperature above the optimum (5°C to 28°C), it is estimated that kernel number decreases by 4% (NSW DPI, 2007). An extreme heat event at grain fill can negatively impact kernel size as it disrupts the translocation of nutrients and the synthesis and deposition of starch.

The most effective way of assessing the performance of a genotype and its range of physiological responses is to screen materials under stressful environmental conditions. Screening methods for heat tolerance often involve single tiered techniques, usually in a glasshouse setting, which can lack relevance to farming environments and does not always represent the most relevant materials. This paper discusses the findings of the second tier of a three-tiered technique developed specifically to screen for heat tolerance (Thistlethwaite et al., 2020). This method evaluates germplasm by combining natural and controlled conditions using in-field controlled environment chambers.

Methods

Twenty-three current commercial bread wheat cultivars and two breeding lines were screened under in-field controlled environment chamber conditions at The University of Sydney Plant Breeding Institute, Narrabri in 2020. Each line was sown in 12 m^2 replicated plots. Treatments involved a heated chamber that adjusted its internal conditions to be consistently 4°C above the ambient temperature. Anthesis treatments consisted of an average increased temperature of 30.7°C between 10:00-14:00h and grain fill treatments consisted of an average increased temperature of

15.3°C between 19:00-07:00h. Chamber units were deployed at the same growth stage at the same time of day for each genotype.

The hidden costs associated with a rise in global minimum temperatures, especially late in the season, is the reason a nighttime heat treatment was imposed at grain fill. Different physiological processes occur within a plant during the night compared with during the day. Some varieties could be more efficient at recovering during the night which would then allow them to cope much better when the temperature was high during the day. The physiological basis of this phenomenon remains limited; however, evaluations such as this are a step towards a better understanding of why, rather than how, varieties differ from each other.

In-field controlled environment chambers, designed to fit over a 4 m^2 of a plot, were used to increase the maximum ambient temperature for three consecutive days at anthesis and increase the minimum ambient temperature for three consecutive nights during grain fill (Figure 1). Each chamber unit was 2 m x 2 m x 1.5 m (length x width x height) in dimension. Chamber units consisted of a galvanised steel frame covered in clear polycarbonate. HOBOware® weatherproof temperature and humidity data loggers with a temperature measurement range of -40 °C to 70 °C with ±0.2 °C accuracy and a relative humidity (RH) measurement range of 0 to 100% with ±2.5% accuracy was installed in all units and fitted with a radiation shield.

Figure 1. In-field controlled environment chambers in the field at The University of Sydney Plant Breeding Institute, Narrabri 2020.

A positive temperature coefficient (PTC) thermistor with resistance increasing with temperature was installed with output fed to a SPLat HVAC26A AC controller. Each individual chamber was fitted with a domestic reverse cycle air conditioning (AC) unit rated to 3.9 kW heating and 3.6 kW cooling with a maximum full load current of 8.5 amps and ducting ran from the AC unit to the chamber. A 15 kVA 3-phase diesel generator was used to provide in-field power to the chamber units.

Spikes that were deemed to be at the correct stage for treatment were tagged and subsequently harvested to ensure the accuracy of plant growth stage. Yield, yield components and targeted phenological and physiological traits were assessed at key growth stages. Irrigation was applied based on Goanna moisture (capacitance) probe readings to ensure well-watered conditions throughout the season.

Ideally, this evaluation of commercial varieties should be repeated across multiple years and sites due to potential variation in environmental conditions and to allow for the best representation of the germplasm. In previous years, in-field controlled environment chambers were used primarily to assess new heat tolerant material rather than commercial varieties due to the availability of chambers.

Results

Overall, in-field controlled environment chambers significantly impacted the yield and kernel weight of most lines evaluated when a heat shock was applied at anthesis and during grain fill (Table 1). An average decrease in yield of 14% and kernel weight of 9.7% was observed between control and heated plots when a heat shock was applied during the day at anthesis. When minimum temperatures were increased at night during grain fill, yield and kernel were not as greatly impacted with yield and kernel weight decreases of 4.8% and 5.8% observed, respectively.

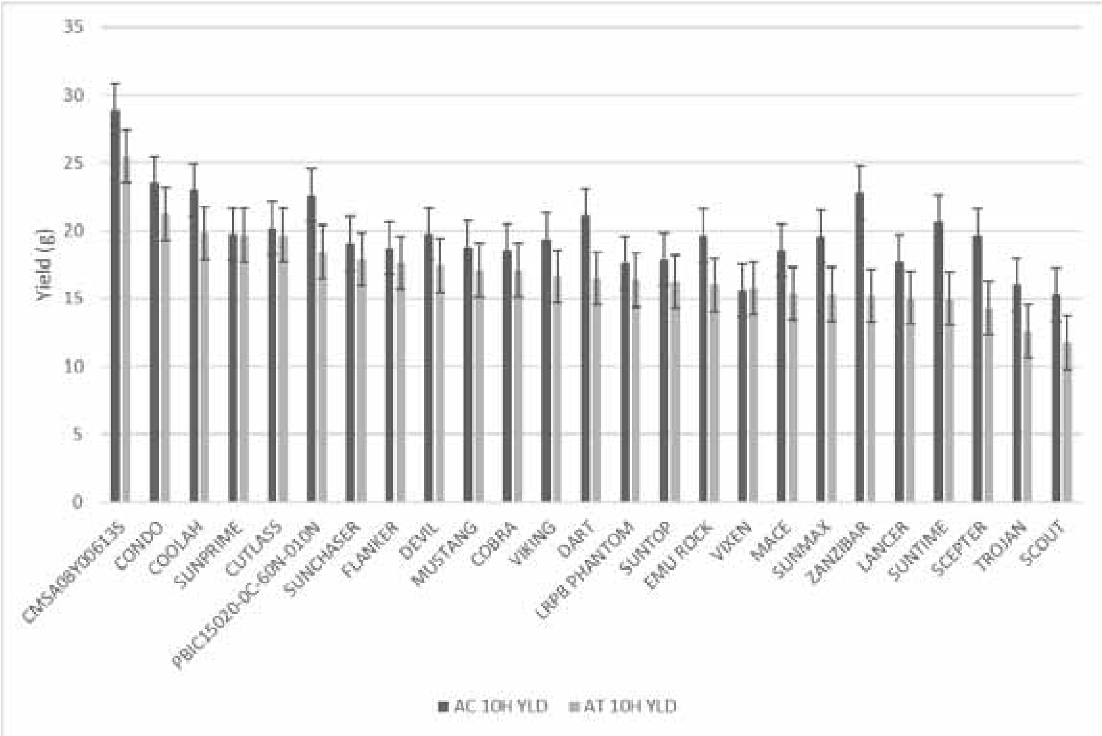
Table 1. The impact of heat chambers on yield and kernel weight (TKW) of ten heads selected at anthesis (Zadok 60) and mid-grain fill (Zadok 73) respectively.

Trait	Anthesis Control	Anthesis Treated	Grain Fill Control	Grain Fill Treated	Prob.
Yield of 10 heads (g)	19.78 a	16.95 b	19.28 a	18.35 c	<0.001
TKW of 10 heads (g)	38.28 a	34.57 b	37.86 a	35.67 b	<0.001

Note: means in rows followed by the same letter are not significantly different at P<0.05

Recent Australian varieties performed well under anthesis heat treatment with the best performing varieties AGT Condo, Coolah, Sunprime and Cutlass combining yield potential and heat tolerance, closely followed by AGT Sunchaser, LRPB Flanker and Intergrain Devil. Varieties such as LRPB Dart, RGT Zanzibar and AGT Scepter showed good yield potential and performed well under controlled conditions but were greatly affected by heat treatment. Intergrain Vixen seemed to be unaffected by heat treatment as the difference between control and treatment yield was negligible. CIMMYT breeding line (CMSA08Y00613S-050Y-050ZTM-050Y-59BMX-010Y-0B) significantly outperformed all current varieties for yield (56% better than AGT Suntop) and kernel weight under control and heat treatments (Figure 3).

(a)



(b)

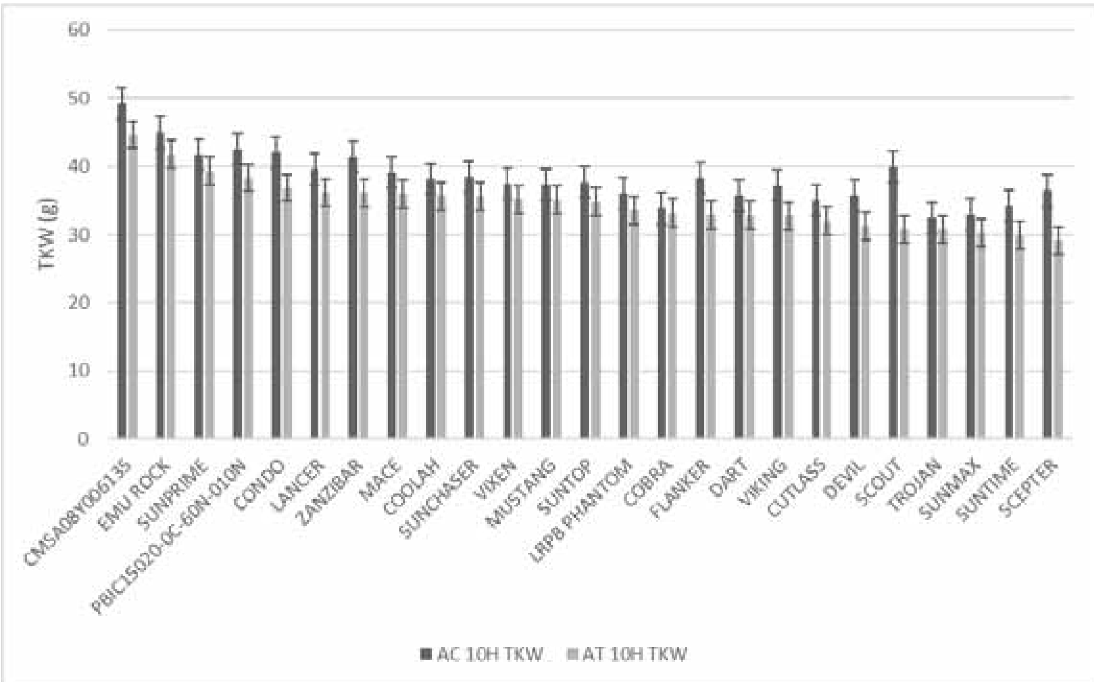


Figure 2. Yield (a) LSD ≤0.05 =3.98 (AC 10H YLD - Anthesis control 10 head yield, AT 10H YLD – Anthesis treatment 10 head yield) and kernel weight (b) LSD ≤0.05 =4.42 (AC 10H TKW - anthesis control 10 head thousand kernel weight, AT 10H TKW - anthesis treatment 10 head thousand kernel weight) of Australian cultivars under elevated day temperatures treatment and associated control at anthesis, Narrabri 2020.

(All the named varieties in the graphs above are protected under the Plant Breeders Rights Act 1994)

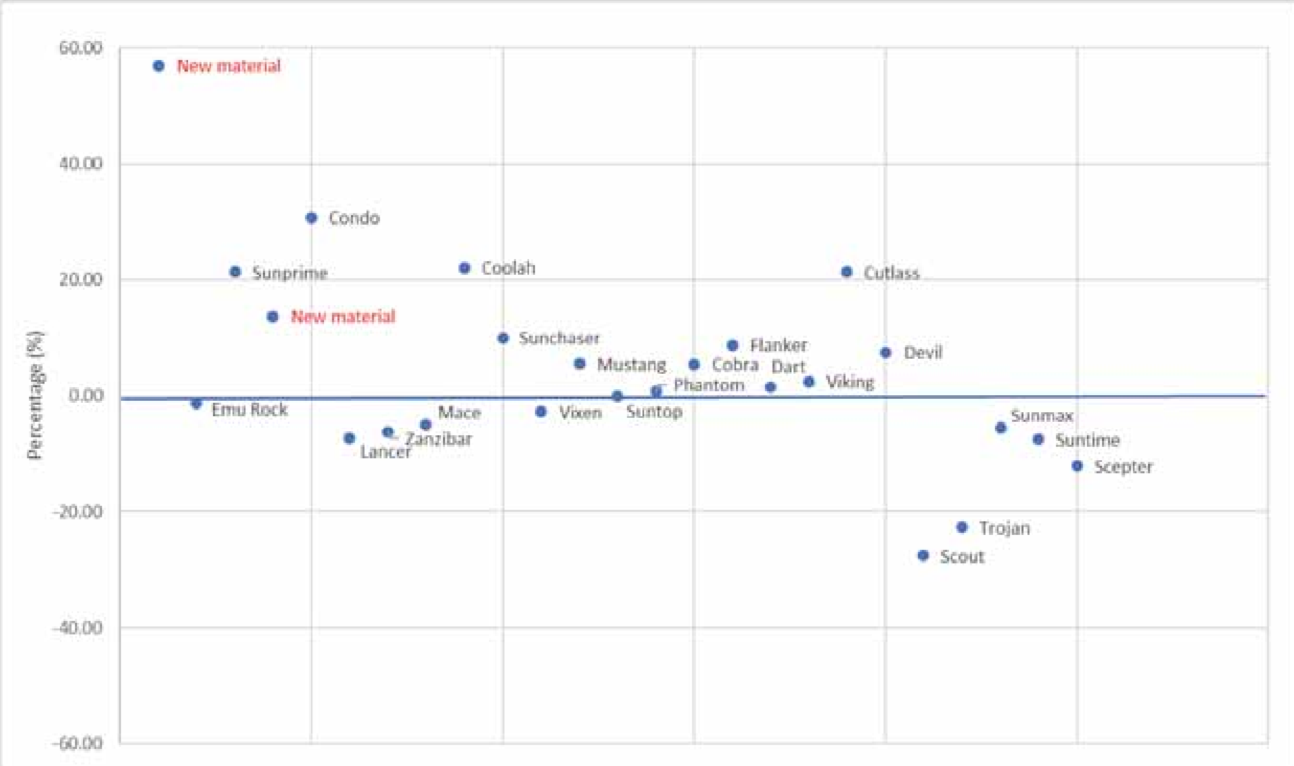
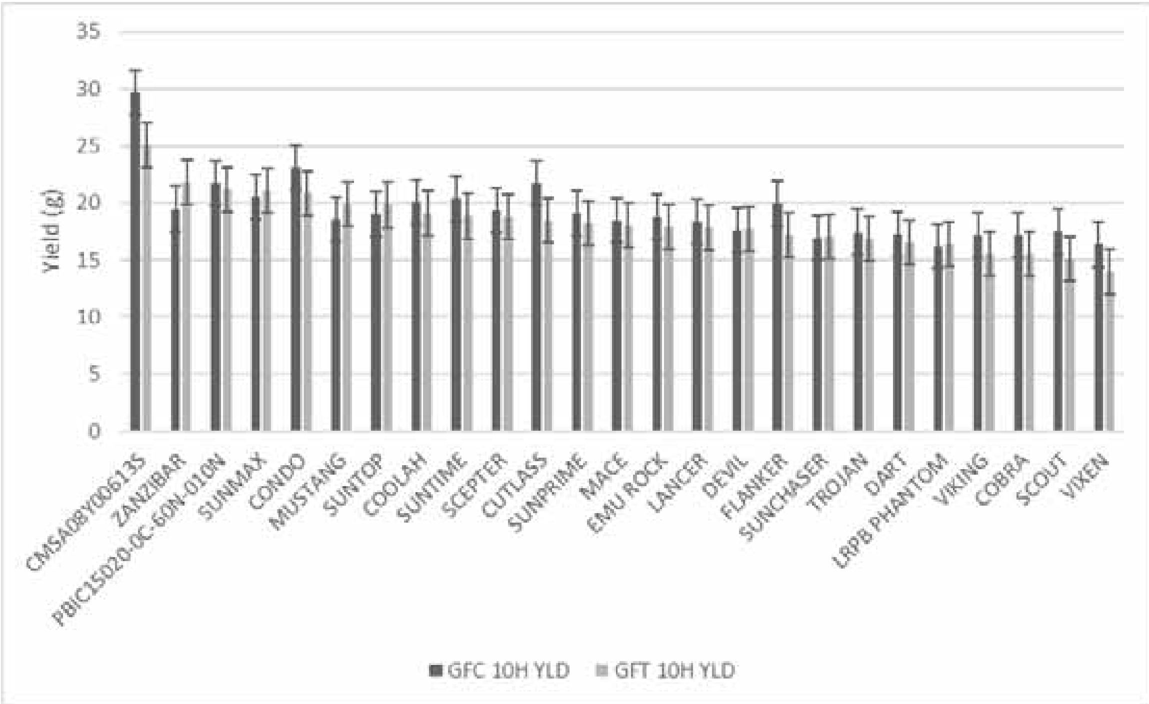


Figure 3. Yield of 10 heads for anthesis treatment of current cultivars under elevated day temperature treatment as a percentage (%) of Suntop⊕ which is represented by the x-axis (0.00%). (All the named varieties in the graphs above are protected under the Plant Breeders Rights Act 1994)

This was closely followed by AGT Suntop⊕, Coolah⊕, Suntime⊕ and Scepter⊕ (Figure 4). In some cases, as with RGT Zanzibar⊕, AGT Sunmax⊕ and Suntop⊕ and LRPB Mustang⊕, the heat treatment slightly outperformed the control. The varieties that were the most stable between treatments included Intergrain Devil⊕, AGT Sunchaser⊕ and LRPB Phantom⊕ although they didn't have as high a yield potential. CIMMYT breeding line (CMSA08Y00613S-050Y-050ZTM-050Y-59BMX-010Y-0B) surpassed all current varieties for kernel weight.

(a)



(b)

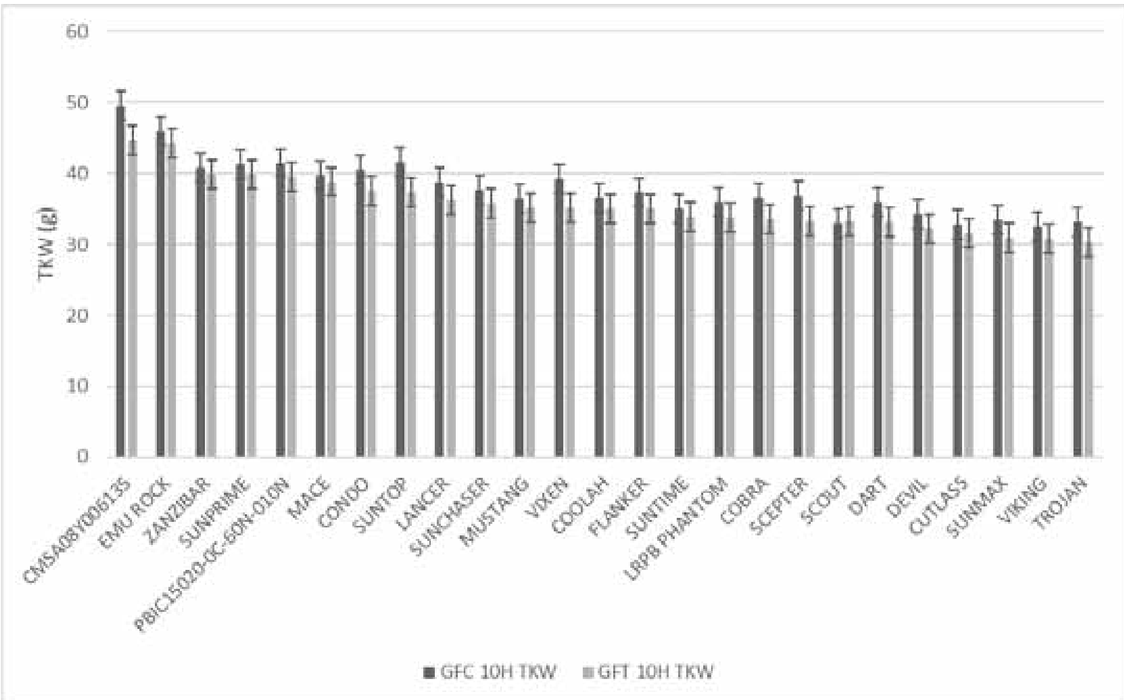


Figure 4. Yield (a) LSD ≤0.05 = 3.57 (GFC 10H YLD – grain fill control 10 head yield, GFT 10H YLD – grain fill treatment 10 head yield) and kernel weight (b) LSD ≤0.05 = 3.81 (GFC 10H TKW – grain fill control 10 head thousand kernel weight, GFT 10H TKW – grain fill treatment 10 head thousand kernel weight) of Australian cultivars under elevated minimum ambient temperatures treatment and associated control at grain fill, Narrabri 2020.

(All the named varieties in the graphs above are protected under the Plant Breeders Rights Act 1994)

Conclusion

Current and emerging Australian cultivars have a good degree of heat tolerance, however there is still more that can be achieved. This research identified and developed new sources of heat tolerance that can be used by wheat breeders to improve yield in our increasingly variable climate.

References

Jozef Syktus, Nathan Toombs, David Ahrens, Kenneth Koon-Ho Wong, Ramona Dalla Pozza, Heatwaves intensification in Australia: A consistent trajectory across past, present and future, Science of The Total Environment, Volume 742, 2020, https://doi.org/10.1016/j.scitotenv.2020.140521.

NSWDPI, 2007. Wheat Growth and Development. In: White, J., Edwards, J. (Eds.), http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0008/516185/Procrop-wheat-growth-and-development.pdf, p. 93.

Rebecca. J. Thistlethwaite, Daniel. K.Y. Tan, Anowarul. I. Bokshi, Smi Ullah, Richard. M. Trethowan, A phenotyping strategy for evaluating the high-temperature tolerance of wheat, Field Crops Research, Volume 255, 2020, 107905, ISSN 0378-4290, https://doi.org/10.1016/j.fcr.2020.107905.

Acknowledgements

The authors acknowledge the funding support of the GRDC, the University of Sydney and Agriculture Victoria, and the in-kind contributions of Intergrain and AGT.

Contact details

Rebecca Thistlethwaite
University of Sydney
Ph: 0403 525 345
Email: rebecca.thistlethwaite@sydney.edu.au

⦶ Varieties displaying this symbol beside them are protected under the Plant Breeders Rights Act 1994.

® Registered trademark

NSW cereal diagnostics and enquiries – the 2020 winner is……?

Steven Simpfendorfer[1], Brad Baxter[2] and Andrew Milgate[2]

[1] *NSW DPI Tamworth*
[2] *NSW DPI Wagga Wagga*

Keywords

correct diagnosis, leaf diseases, soil-borne diseases, wheat, barley

GRDC code

DAN00213: Grains Agronomy & Pathology Partnership (GAPP) - A strategic partnership between GRDC and NSW DPI. Projects BLG208 and BLG207.

Take home messages

- Cereal diseases were prevalent in 2020 with favourable climatic conditions. Hence, in combination with increased cereal stubble loads, pathogen levels are likely to be elevated in 2021
- However, steps can be taken to minimise impacts which include:
  - Remember the basics of disease management – think disease triangle!
  - Know before you sow (e.g. PREDICTA®B or stubble tests) – inoculum levels
  - Varietal resistance – reduce host susceptibility
  - Manipulate canopy microclimate or stress during grain filling – environmental conditions
- NSW DPI plant pathologists can help with correct diagnosis and management options.

Introduction

A ‘no-additional charge’ cereal diagnostic service is provided to NSW cereal growers and their advisers under projects BLG207 and BLG208 as part of the GAPP co-investment. Evidence based methods are used to confirm diagnosis which include a combination of visual symptoms, crop management history, distribution in paddock and recovery/identification of the causal pathogens (microscopy, humid chamber or plating). Any suspect virus samples are confirmed using ELISA antibody testing at NSW DPI Elizabeth Macarthur Agricultural Institute at Menangle.

Wheat, barley and oat rust samples (stripe, leaf and stem) are sent to the Australia Cereal Rust Control Program (ACRCP). The submission of samples to ACRCP facilitates the tracking of pathotype populations and distribution across the cropping belt of NSW and Australia. This includes a new interactive map (Australian Cereal Rust Survey 2020 Sample Map - Google My Maps) regularly updated throughout the growing season by the ACRCP. Growers can access this resource to see which pathotypes dominate in their region. This can be very important to guide in-crop management decisions given five different stripe rust pathotypes were present at varying levels across NSW in 2020. Individual wheat varieties can have vastly different reactions to these pathotypes, so knowing which ones are dominant, where and when, can guide appropriate seasonal in-crop management.

The projects also record disease enquiries received and resulting management advice provided to growers and advisers throughout each season. These project activities support NSW cereal producers in correct diagnosis of diseases during the season with resulting independent advice on appropriate management strategies to limit economic impacts. This is assisting to limit the unnecessary application of in-crop fungicides by growers.

Which diseases dominated in 2019 and 2020?

Collation of this data across NSW provides an annual 'snapshot' of the key biotic and abiotic constraints to cereal production (Table 1).

Table 1. Cereal diagnostics and enquiries processed across NSW in 2019 and 2020. Disease/issues are ranked in order of frequency in 2020

Disease/issue	2020	2019
Stripe rust (wheat)	194	13
Spot form of net blotch	65	32
Physiological/melanism	65	10
Scald	65	4
Fusarium crown rot	61	14
Wheat powdery mildew	53	1
Frost damage	45	4
Leaf rust (wheat)	35	2
Other non-disease (e.g. soil constraint, leaf blotching)	34	24
Bacterial blight (other cereals)	30	0
Rusts crown and stem (oats)	29	4
Herbicide	28	6
Net form of net blotch	23	0
Bacterial blight (oats)	22	3
Barley grass stripe rust	20	1
Barley yellow dwarf virus	19	1
Septoria tritici blotch	17	13
Nutrition	16	2
Take-all	16	1
Rhizoctonia	12	7
Barley powdery mildew	12	0
Yellow leaf spot	10	4
Fusarium head blight	10	0
Loose smut	9	1
Seedling root disease complex (Pythium, crown rot, Rhizo,Take-all)	8	2
Septoria oats	3	2
Wheat streak mosaic virus	3	1
Common root rot	2	3
Rye grass rust	2	0
Ergot	1	0
Red leather leaf	1	7
Sclerotium rolfsii	1	2
Spot blotch	1	0
Ring spot	0	1
Total	**912**	**165**

Not surprisingly, individual seasons have a strong influence on the level of cereal diagnostic support provided to NSW growers/advisers, with over five-times the number of activities in the wetter 2020 season compared with much drier conditions experienced in 2019 (Table 1). This increase was primarily due to more conducive conditions for the development of a range of cereal leaf diseases (e.g. rusts, scald, net-blotches, Septoria) in 2020 (537 samples) compared with 2019 (77 samples).

The four main cereal diseases in 2020 were wheat stripe rust (widespread distribution of newer Yr198 pathotype), spot form of net blotch in barley, scald in barley and Fusarium crown rot in different winter cereal crops. In comparison, the four main cereal diseases in 2019 were spot form of net blotch, Fusarium crown rot, wheat stripe rust and Septoria tritici blotch (Table 1).

Interestingly, the levels of yellow leaf spot (*Pyrenophora tritici-repentis*) diagnosed in both seasons were relatively low. However, wheat samples with leaf blotches or mottling were submitted each year, suspected to be caused by yellow leaf spot. There is an ongoing difficulty with correct diagnosis of this particular leaf disease by growers and their advisers, often confused with Septoria tritici blotch (*Zymoseptoria tritici*), Septoria nodorum blotch (*Stagonospora nodorum*) and physiological responses to abiotic stress (e.g. frost yellowing, N mobilisation, herbicide damage).

The 2020 season also highlighted that root diseases like take-all, which have not been seen at damaging levels for many years can quickly re-emerge at significant levels when conducive conditions occur. Conversely, Fusarium crown rot remains a significant issue across seasons.

The number of rust and powdery mildew samples received from susceptible wheat varieties in 2020 highlights the importance of genetic resistance as a component of integrated disease management systems. Susceptible varieties are more reliant on fungicide applications to limit disease levels and associated yield loss, which can increase the risk of fungicide resistance developing. The resistance selected may not necessarily be in the main pathogen targeted by the fungicide applications. For example, reliance on fungicide applications in stripe rust susceptible varieties could inadvertently select for fungicide resistance in wheat powdery mildew populations when they co-infect plants. Preliminary research conducted in collaboration with Curtin University's Centre for Crop Disease Management (CCDM) in 2020, unfortunately indicates issues with reduced sensitivity to azoles (DMIs, Group 3) and resistance to strobilurin (QoIs, Group 11) fungicides are already widespread in wheat powdery mildew populations in NSW and Victoria.

Are you getting a correct diagnosis?

Importantly, 21% of activities in 2020 and 28% in 2019 were not related to disease. These samples were either diagnosed as being plant physiological responses to stress, frost damage, herbicide injury, related to crop nutritional issues or other non-disease issues. All of these samples were submitted as suspected of having disease issues. This highlights the ongoing importance of the diagnostic service provided by these projects to NSW growers and their advisers to support correct identification and implementation of appropriate management strategies. Never be afraid to get a second opinion from a plant pathologist, we are here to help (see contact details).

Management in 2021 – remember the basics!

Showing our ages here by referring back to the good old 'disease triangle', my mentors would be proud! Disease levels in 2021 will still be based around the disease triangle, which requires a combination of pathogen inoculum, susceptible host and environmental conditions conducive to disease development. Given the elevated incidence of a wide range of cereal diseases across NSW in 2020 (Table 1), inoculum levels of a range of cereal pathogens and hence disease risk in 2021 will be higher than previous seasons. Each of the three components of the disease triangle should be considered when implementing management strategies to minimise losses and determine if fungicide application is warranted in 2021.

1. Inoculum levels

The first step is 'know before you sow'. PREDICTA®B testing remains the gold standard for a quantitative assessment of a wide range of cereal pathogens and associated risk of both soil-borne and leaf diseases. Refer to [the PREDICTA B sampling protocol (https://pir.sa.gov.au/__data/assets/pdf_file/0007/291247/Sampling_protocol_PreDicta_B_Northern_regions.pdf)]. NSW DPI is alternatively offering a free cereal stubble testing service prior to sowing in 2021 (Jan-Apr) aimed primarily at determining Fusarium crown rot risk levels in cereal-on-cereal situations (contact Steven Simpfendorfer for details).

The disease risk associated with inoculum levels can be quite different with various pathogens depending on their capacity for wind dispersal. For example, stubble and soil borne pathogens which cause Fusarium crown rot, take-all and Rhizoctonia root rot are not dispersed by wind, hence risk from inoculum is confined to an individual paddock. Consequently, crop rotation to a non-host pulse or oilseed crop breaks the disease triangle. Stubble borne leaf pathogens, which cause net blotch or scald in barley, yellow spot or Septoria tritici blotch in wheat or powdery mildew, have limited wind dispersal (i.e. metres), so again crop rotation largely reduces disease risk and especially at early growth stages. Conversely, rusts are airborne (i.e. kilometres) so crop rotation is irrelevant to disease risk.

Seed borne infection should also be considered with some pathogens such as bacterial blights, scald, net-form of net blotch, smuts and bunts. Sourcing clean seed for sowing in 2021, that is, not from crops infected in 2020, is important to reduce risk of these diseases.

With stripe rust, reducing or delaying the onset of an epidemic significantly reduces disease pressure. Rust spores are readily wind borne and are commonly referred to as 'social diseases' (i.e. 'we are all in this together'). Hence, co-ordinated management across a region can have real benefits for all. Controlling volunteer wheat plants at least three weeks prior to first planting of crops limits the 'green bridge' survival and delays epidemic onset. In-furrow (e.g. flutriafol) and seed treatments (e.g. fluquinconazole) fungicides provide extended protection from stripe rust early in the season delaying epidemic onset. This can be particularly important when early sowing susceptible long season wheat varieties (e.g. DS Bennett), which can place early disease pressure on later sown susceptible main season varieties.

Growers should also be aware that stubble management practices can also influence inoculum dispersal. For example, inter-row sowing between intact standing cereal stubble reduces the level of Fusarium crown rot infection. However, cultivating or mulching infected cereal stubble prior to sowing can spread Fusarium inoculum more evenly across a paddock and potentially into the surface layers of the soil where plant infection primarily occurs. Volunteer cereal plants and grass weeds over the summer fallow period can also be a major source of increased inoculum of Fusarium crown rot, take-all and Rhizoctonia leading into sowing in 2021.

2. Host susceptibility

Relatively self-explanatory? If you do not want cereal disease issues, then sow a non-host pulse or oilseed break crop. However, if considering cereal-on-cereal, key points are:

1. Make sure you are using the latest varietal resistance ratings especially to newer pathotypes of stripe rust. Many growers got caught out on this with durum wheats and DS Bennett in 2020. Improved levels of resistance to leaf diseases reduces reliance on foliar fungicides
2. If multiple pathogen risks then hedge towards improved resistance to the more yield limiting, harder to control and/or historically bigger issue in your area. This could be quite different between rainfall zones or dryland vs irrigated situations

3. Barley, bread wheat, durum, oats and triticale are NOT break crops for each other! They all host Fusarium crown rot, take-all and Rhizoctonia. Barley tends to be more susceptible to Rhizoctonia root rot, but its earlier maturity can provide an escape from late season stress which reduces yield loss to Fusarium crown rot
4. Rusts and necrotrophic leaf diseases (net blotches, yellow spot, Septoria) tend to be crop specific. However, note that in wetter seasonal finishes it appears that some of these necrotrophic leaf pathogens have the potential to saprophytically colonise other cereal species.

3. Environmental conditions

Largely in the 'lap of the gods'? Certainly more limited options here but growers should be aware that subtle microclimate differences within cereal canopies can have a large influence on cycling of leaf diseases. Crash grazing of dual purpose wheat varieties not only reduces any early stripe rust inoculum, but also opens the canopy to reduce the duration of leaf wetness and lowers humidity. This reduces conduciveness to leaf diseases. Higher nitrogen levels can also exacerbate rusts and powdery mildew through thicker canopies creating a more favourable microclimate for these pathogens. However, leaf nitrate also serves as a food source for these biotrophic leaf pathogens.

Yield loss from Fusarium crown rot infection, largely through the expression of the disease as whiteheads, is strongly related to moisture and temperature stress during grain filling. Although growers cannot control rainfall during this period, there is the potential to limit the probability of stress through earlier sowing (matched to varietal maturity and frost risk), maximising soil water storage during fallow periods (stubble cover + weed control), addressing other biotic (e.g. nematodes, Rhizoctonia) or abiotic (e.g. acidity, nutrition, residual herbicides etc.) constraints to root development and canopy management. Recent (last two weeks) and predicted weather conditions (next 2-4 weeks) should also be considered with in-crop leaf disease management decisions in susceptible varieties around key growth stages for fungicide application of GS30-32 (1st node), GS39 (flag leaf emergence) and GS61 (flowering).

Conclusions

Overall the 2020 season was fairly good across a large proportion of NSW with cereal diseases present at higher frequencies than recent seasons. Hopefully 2021 provides another favourable year for cereal production. Cereal disease risk is likely to be higher due to pathogen build-up in 2020 and the likely increased area of cereal-on-cereal in 2021. Calm considered and well planned management strategies in 2021 can minimise disease levels. NSW DPI is here to support correct diagnosis and discuss management options prior to sowing and as required throughout the season. Let's get back to cereal disease management basics in 2021 and leave any lingering 'pandemic panic' from 2020 behind.

Acknowledgements

The research undertaken as part of this project is made possible by the significant contributions of growers and their advisers through both sample submission and the support of the GRDC, the author would like to thank them for their continued support. The author would also like to acknowledge the ongoing support for northern pathology capacity by NSW DPI.

Contact details

Steven Simpfendorfer
NSW DPI, 4 Marsden Park Rd, Tamworth, NSW 2340
Ph: 0439 581 672
Email: steven.simpfendorfer@dpi.nsw.gov.au
Twitter: @s_simpfendorfer or @NSWDPI_AGRONOMY

Andrew Milgate
NSW DPI, Pine Gully Rd, Wagga Wagga, NSW 2650
Ph: 02 6938 1990
Email: andrew.milgate@dpi.nsw.gov.au

® Registered trademark.

KEY CONTACTS

GRDC
GRAINS RESEARCH & DEVELOPMENT CORPORATION

NORTHERN REGION

TOOWOOMBA
214 Herries Street
TOOWOOMBA, QLD 4350

northern@grdc.com.au
P:+61 7 4571 4800

OPERATIONS GROUP

SENIOR REGIONAL MANAGER
Gillian Meppem
Gillian.Meppem@grdc.com.au
M: +61 4 0927 9328

BUSINESS SUPPORT TEAM LEADER
Adam van Genderen
Adam.vangenderen@grdc.com.au
M: +61 4 2798 4506

CONTRACT ADMINISTRATOR AND PANEL SUPPORT
Tegan Slade
Tegan.Slade@grdc.com.au
M: +61 4 2728 9783

CONTRACT AND TEAM ADMINISTRATOR
Brianna Oram
Brianna.Oram@grdc.com.au
P: +61 7 4571 4800

APPLIED RESEARCH, DEVELOPMENT AND EXTENSION GROUP

GENERAL MANAGER APPLIED RD&E (NATIONAL)
Peter Carberry
Peter.Carberry@grdc.com.au
M: +61 4 1965 6955

SENIOR MANAGER CROP PROTECTION (NATIONAL)
Emma Colson
Emma.Colson@grdc.com.au
M: +61 4 5595 8283

SENIOR MANAGER EXTENSION AND COMMUNICATION (NATIONAL)
Luke Gaynor
Luke.Gaynor@grdc.com.au
M: +61 4 3666 5367

SENIOR MANAGER AGRONOMY, SOILS, NUTRITION AND FARMING SYSTEMS (NATIONAL)
Michael Bange
Michael.Bange@grdc.com.au
M: +61 4 4876 6881

CROP PROTECTION MANAGER
Vicki Green
Vicki.Green@grdc.com.au
M: +61 4 2904 6007

GROWER RELATIONS MANAGER
Richard Holzknecht
Richard.Holzknecht@grdc.com.au
M: +61 4 0877 3865

MANAGER AGRONOMY, SOILS, NUTRITION AND FARMING SYSTEMS
Kaara Klepper
Kaara.Klepper@grdc.com.au
M: +61 4 7774 2926

MANAGER CHEMICAL REGULATION (NATIONAL)
Gordon Cumming
Gordon.Cumming@grdc.com.au
M: +61 4 2863 7642

GROWER RELATIONS MANAGER
Graeme Sandral
Graeme.sandral@grdc.com.au
M: +61 4 0922 6235

MANAGER AGRONOMY, SOILS, NUTRITION AND FARMING SYSTEMS
John Rochecouste
John.Rochecouste@grdc.com.au
M: +61 4 7774 2924

CONTRACT ADMINISTRATOR
Linda McDougall
Linda.McDougall@grdc.com.au
M: +61 4 7283 2502

GENETICS AND ENABLING TECHNOLOGIES GROUP

NATIONAL VARIETY TRIALS OFFICER
Laurie Fitzgerald
Laurie.Fitzgerald@grdc.com.au
M: +61 4 5595 7712

COMMUNICATIONS GROUP

COMMUNICATIONS MANAGER
Toni Somes
Toni.Somes@grdc.com.au
M: +61 4 3662 2645

BUSINESS AND COMMERCIAL GROUP

MANAGER COMMERCIALISATION
Chris Murphy
Chris.Murphy@grdc.com.au
M: +61 4 2277 2070