

Integrated Pest Management Guidelines for Cotton Production Systems in Australia



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Acknowledgements

We thank the following people and their organisations for their assistance with providing and revising the text, and general advice on the booklet layout. We would also like to acknowledge the contributors to the first IPM guidelines.

DPI&F, Queensland: Dr Melina Miles, Mr Mark Hickman, Dr Mark Wade, Dr David Murray, Ms Ingrid Christianson, Dr Richard Sequeria, Dr Austin McLennan, Dr Moazzem Khan, Dr Brad Scholtz and Mr Richard Lloyd.

NSW DPI: Dr Robert Mensah, Mr David Larsen, Mr Dallas Gibb, Mrs Annie Johnson, Dr Louise Rossiter, Dr Grant Herron, Dr Robin Gunning and Mr Viliami Heimoana.

CSIRO Plant Industry: Ms Sandra Deutscher, Dr Lewis Wilson, Dr Michael Bange and Dr Greg Constable.

CSIRO Entomology: Mr Martin Dillon, Mr Colin Tann and Dr Gary Fitt.

CRDC: Mr Bruce Pyke and Mr Greg Kauter.

CSD: Mr David Kelly (also representing the CRC Insect extension focus team).

ACGRA: Mr Allan Williams, Mr Jeff Bidstrup and Mr Peter Glennie.

CCA: Mr Iain MacPherson, Mr Rick Thomas, Mr John O'Brien and Mr Chris Lehmann.

Bayer CropScience: Dr Jonathan Holloway (formerly NSW DPI).

Delta Pine and Land Copmpany: Dr Neil Forrester (formerly NSW DPI).

Thank you to the following people who have provided images: Mr Martin Dillon, Ms Cheryl Mares, Dr Neil Forrester, Mr David Kelly, Dr Ian Rochester, Dr Brad Scholtz, L Turton, M Cahill, Dr Peter Room, D Ironside, Mr David McClenaghan, Dr Robert Mensah, Dr Graham Charles and Mrs Julie Ferguson.

Thanks are also due to Gillian Stack for her skills and patience with the design and layout of the guidelines.

Sources of information

Many of the information packages have titles which end in 'pak'. For example, WEEDpak, ENTOpak, NUTRIpak, MACHINEpak, WATERpak, SPRAYpak and DISEASEpak. The majority of these packages are delivered in a folder format, although all of the 'paks' are now being delivered on a 'Cottonpaks' CD for more efficient delivery. There are a number of research reviews published on specific issues, such as mite ecology, aphid management, whiteflies, and so on, which are referred to in the IPM guidelines. All information paks and research reviews are available from the Australian Cotton CRC Technology Resource Centre (TRC) and also on the Australian Cotton CRC website (cotton.crc.org.au). The coordinator of the TRC can be contacted on 02 6799 1534.

Two other key tools are also available on the Australian Cotton CRC website. These are the '*Cotton Pest Management Guide*', which details all of the insecticides available for use in cotton, and the '*Pest and Beneficial Guide*', which provides detailed information and pictures for the range of pests and beneficials found on cotton.

The computerised decision support packages developed by the CSIRO and the Australian Cotton Cooperative Research Centre, are identified by the term 'LOGIC' as they belong to the suite of CottonLOGIC tools. For example, HydroLOGIC, NutriLOGIC and EntomoLOGIC. The software is available from the TRC and the latest software upgrades can be downloaded from the Australian Cotton CRC website.

Disclaimer

The 'Integrated Pest Management Guidelines for Australian Cotton Production Systems', 2nd edition (IPM guidelines) is designed to be used as a tool to help improve pest management in cotton. It is not a substitute for personnel with expert knowledge of pest management or any other aspects of cotton crop management.

The IPM guidelines is made available to the cotton industry in anticipation of feedback from the users regarding potential improvements or problems encountered. This process ensures continuing development and improvement of the guidelines.

The Australian Cotton CRC and its core partners do not warrant or make any representation regarding the use of, or the results of the use of, the IPM guidelines. In particular the Australian Cotton CRC and its core partners do not warrant or represent that any of the management strategies within the IPM guidelines are correct, accurate or reliable. The user relies on the guidelines at their own risk.

IMPORTANT: USE OF PESTICIDES

Pesticides must only be used for the purpose for which they are registered and must not be used in any other situation or in any manner contrary to the directions on the label.

Some chemical products have more than one retail name. All retail products containing the same chemical may not be registered for use on the same crops. Registration may also vary between States. Check carefully that the label on the retail product carries information on the crop to be sprayed.

This publication is only a guide to the use of pesticides. The correct choice of chemical, selection of rate, and method of application is the responsibility of the user.

Pesticides may contaminate the environment. When spraying, care must be taken to avoid spray drift on to adjoining land or waterways. Residues may accumulate in animals fed any crop product, including crop residues, which have been sprayed with pesticides. In the absence of any specified grazing withholding period(s), grazing of any treated crop is at the owner's risk.

Definitions

Throughout these guidelines the term 'Bollgard II® refuge' is used to refer to crops grown specifically as a requirement of the Bollgard II® licence to produce *Bacillus thuringiensis* (BT) susceptible *Helicoverpa* spp. The term 'beneficial insect refuge' is used to refer to crops planted specifically as a source of refuge for beneficial insects and spiders.

Sprays that could be applied for pest management are defined as food sprays, selective biological's, selective synthetic insecticides and broad-spectrum synthetic insecticides. Selective insecticides are those with a low negative effect on beneficials, while broad-spectrum insecticides are those that have a high negative effect on beneficials. Many insecticides fall in between these two ends of the spectrum.

Integrated Pest Management Guidelines for Cotton Production Systems in Australia

IPM Guidelines 2nd Edition

These guidelines have been designed to assist growers implement IPM strategies to suit their individual farming system.

The original IPM guidelines were developed in response to requests from growers and consultants for information to work with when implementing an IPM program.

The original version was a collaborative effort between growers and consultants attempting to practice IPM, researchers and extension officers.

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1. What is Integrated Pest Management?

1.1 Introduction

1.1.1 The definition of Integrated Pest Management?

Integrated Pest Management (IPM) involves using all means of managing pest populations with the aim of reducing insecticide use while maintaining profitability, yield and fibre quality. IPM is a whole year approach to managing pests. This includes management of pests through the cotton growing season, and through the remainder of the year as well. For instance, decisions made in the autumn and winter can have a lasting impact on pest management throughout the year.

An operational definition of IPM developed by the Food and Agriculture Organisation is:

- the presence of pests does not automatically require control measures, as damage may not be significant
- when pest control measures are deemed necessary, a system of non-chemical pest methodologies should be considered before a decision is taken to use pesticides
- a suitable pest control strategy should be used in an integrated manner and pesticides should be used appropriately
- intervention with broad spectrum synthetic pesticides is seen as a last resort when pests exceed thresholds and there are no effective selective management options available.

1.1.2 Why do we need to develop IPM programs?

Cotton crops in Australia are attacked by a wide range of pests, the major ones being *Helicoverpa armigera* (cotton bollworm), *Helicoverpa punctigera* (native budworm), *Creontiades dilutus* (green mirid), *Aphis gossypii* (cotton aphid), *Tetranychus urticae* (two-spotted spider mite) and *Bemisia tabaci* B- Biotype (silverleaf whitefly). Control of these pests has largely relied on the use of synthetic insecticides. Over reliance on synthetic insecticides creates problems, such as insecticide resistance (in *H. armigera*, silverleaf whitefly, aphids and mites), disruption of natural pest enemies, secondary pest outbreaks and environmental consequences. These problems have cast doubt over the long term viability of the traditional insecticide dominated approach to pest management.

In Bollgard II® crops, the need to spray *Helicoverpa* spp. is dramatically reduced. However, these reductions have allowed other pests, formally controlled inadvertently by sprays targeting *Helicoverpa* spp., to emerge as pests. This includes mirids, green vegetable bugs, aphids and jassids. Use of insecticides against these pests may disrupt natural enemies, creating outbreaks of mites, whitefly or aphids. Hence, in both conventional (non-transgenic) and Bollgard II® cotton there remains a strong incentive to use IPM to help reduce reliance on insecticides.





Using a beat sheet to monitor pest and beneficial insect numbers.

A major goal for the cotton industry is to reduce dependence on insecticides while remaining sustainable. This can be achieved by developing an IPM program that minimises insecticide use through integration of a range of pro-active management tactics, especially the conservation and use of natural enemies (predators and parasites) to control pests.

1.1.3 How do we implement IPM?

IPM involves integrating a range of tools and strategies for managing pests. These can be conveniently grouped into seven main objectives which these guidelines are based on:

1. Growing a healthy crop
2. Keeping track of insects and damage
3. Beneficial insects - use them don't abuse them
4. Preventing the development of insecticide resistance
5. Managing crop and weed hosts
6. Using trap crops effectively
7. Supporting IPM through communication and training

It is also important to consider what to do and when, so that these objectives can be planned and achieved. To help this process the year can be divided into five phases that are logical decision points through the year. They are:

Off-season (Winter)	Growing season
1. Post Harvest	3. Planting to 1 flower per metre (first flower)
2. Pre-Planting	4. First flower to 1 open boll per metre
	5. 1 open boll per metre to harvest

These guidelines have a section titled '*Putting IPM into practice*' which lists the actions that require consideration at different times throughout the year. The remainder of the guidelines include more detailed information on the seven main objectives as listed above. This describes the scientific background of each action and the management and planning decisions involved.

1.1.4 Other information about IPM

1.1.4.1 IPM groups/area wide management



IPM groups are a very effective way to help with the uptake of IPM.

Within the cotton industry there are many examples where growers have formed local groups, commonly known as IPM groups or Area Wide Management (AWM) groups. These are generally formed by growers and consultants to help an area progress its IPM program. The groups facilitate this by agreeing on common goals, sharing information and experiences and communicating ideas and solutions to challenges. Groups are an efficient way to share information so that IPM strategies can be implemented on an area wide basis. This approach increases the chances that the actions of growers and their neighbours are complimentary when implementing IPM. In contrast, a grower trying to implement IPM surrounded by neighbours that are not, may have less chance of success because of chemical drift and disruption of beneficial insects. For further information on AWM refer to objective 7 '*Supporting IPM through communication and training*'.

1.1.4.2 IPM and earliness

In conventional cotton IPM aims to optimise earliness through appropriate selection of cotton variety, sowing date, irrigation strategy, nitrogen rate, use of plant growth regulators and sensible pest management. The target of earliness is to reduce the period from sowing to harvest. This may reduce the need for late season control of *Helicoverpa* spp. with expensive, broad spectrum insecticides, thereby reducing the selection pressure for insecticide resistance. Managing for earliness does not imply stringent control of early season pests by excessive use of insecticides. Such an approach may compromise insecticide resistance management and

profitability. Instead, IPM involves preserving and using beneficial insects more effectively, allowing for some compensatory capacity of the crop, monitoring crop damage to ensure that it does not exceed economic levels and managing pests as necessary with appropriate IPM tactics. For further information refer to the section ‘*Optimising earliness*’ in objective 1.

1.1.4.3 IPM and the insecticide resistance management strategy (IRMS)

IPM helps to manage resistance by reducing overall use of synthetic insecticides and hence selection pressure on *H. armigera* and other resistant pests. The insecticide resistance management strategy (IRMS) aims to manage resistance in pests to insecticides. This is critical for conserving the effectiveness of the selective insecticides that are important for IPM. Without the IRMS these insecticides risk being overused, possibly leading to ineffectiveness due to resistance. The IRMS sets limits on insecticides use. The IPM guidelines indicate when pest control is needed and which type of control may be most appropriate, within the IRMS.

IPM is also important for the long term viability of Bollgard II® transgenic cotton. Conservation of beneficial insects, through IPM, helps to control important pests such as aphids, whiteflies, mites and mirids. This reduces the need to control these pests with insecticides, which selects for resistance. Furthermore, an IPM approach helps to avoid creating situations that increase the risk of losses due to these pests, for instance by avoiding rotation crops that host pests. For more information refer to objective 4 ‘*Preventing the development of insecticide resistance*’.

1.1.4.4 IPM, sampling and record keeping

Thorough unbiased sampling is essential for reliable decision making in IPM, whether in conventional or Bollgard II® cotton. The ability to review the season by comparing the costs and profitability of strategies taken on different fields is important for making improvements. The decision support tool, EntomoLOGIC, provides objective sampling systems and also includes the *Helicoverpa* development model, the mite yield loss model, the predator to pest ratios and thresholds for all pests. It also provides excellent record keeping, pest and predator identification and reporting features. For crop nutrition, especially nitrogen, the NutriLOGIC decision support tool provides a rational basis for fertiliser decisions, both pre-planting and through the season. HydroLOGIC provides an irrigation scheduling tool and the capacity to ask ‘what if?’ questions about irrigation decisions. For a free copy of the CottonLOGIC suite of tools, contact the TRC.

For more information on how to monitor a cotton crop for insects and damage, see objective 2 ‘*Keeping track of insects and damage*’ or refer to the ‘*Integrated Pest Management Booklet*’ in the ‘*Australian Cotton Industry Best Management Practices Manual*’.



H. armigera larva feeding on a small cotton boll. Resistance to insecticides in this pest threatens the viability of cotton production.



Reliable information on pest numbers is critical for IPM. Checking should start early in the cotton season.



2. How to use these guidelines

The IPM guidelines focus on the core objectives that make up an IPM system. These objectives are the subject of the following sections of the guidelines. It is also important to know when different operations should occur or be considered. To help we have developed Table 1 '*Putting IPM into practice*', which summarises the major activities for each phase of the crop cycle and the 'off-season'. This can be used to quickly review the operation or issues for that phase of the year, and more detail can then be sought if needed in the following sections.



Figure 1.

IPM is a year round approach to pest management which includes 'off-season' operations and planning as well as crop management during the growing season.

Table 1. Putting IPM into practice

Phases Objectives	Post harvest	Pre-planting	Planting to 1 flower per metre	1 flower per metre to 1 open boll per metre	1 open boll per metre to harvest
1. Growing a healthy crop	Consider the best rotation crop for the situation. Test soil nutrient status to determine fertiliser requirements for cotton crop. Consider potential disease risks.	Seed bed preparation. Field and cotton variety selection. Plan irrigation and crop management strategies.	Consider planting window. Consider at-planting seed treatment, insecticides and other control options which do not disrupt beneficial insect activity. Water management	Monitor crop for • Water management • Nutrient status • Growth control • Pest control	Make final irrigation decisions. Defoliate when crop is mature (monitor NACB). Keep on top of pests by using appropriate control options.
2. Keeping track of insects and damage	Sample cotton stubble for <i>Helicoverpa armigera</i> pupae after harvest.	Assess risk of wireworms, early thrips, mirids, mites and black field earwigs and decide on seed treatments, granular insecticides or in-furrow insecticide sprays.	Sample for pests, beneficials and parasitism rates in cotton as well as spring trap crops. Monitor early season damage. Track pest trends. Use pest thresholds and the predator / beneficial to pest ratio.	Sample for pests, beneficials and parasitism rates. Track pest trends and incorporate parasitism into spray decisions. Monitor fruit load. Use pest thresholds and the predator / beneficial to pest ratio.	Sample for pests, beneficials and parasitism rates in cotton as well as last generation trap crop. Monitor fruit load. Use pest thresholds and the predator / beneficial to pest ratio. Cease pest control at 30-40% bolls open.
3. Beneficial insects – use them don't abuse them	Plant lucerne (strips or block) in autumn. Consider becoming involved in an IPM or AWM group. Discuss spray management plan with neighbours and consultant.	If planning to release <i>Trichogramma</i> during the season, plan to sow other crops e.g. sorghum. Consider growing a diverse habitat to encourage beneficials.	Sample for beneficials and parasitism rates. If chemical control of a pest is required, refer to the beneficial impact table. Keep track of the BDI and predator / beneficial to pest ratio.	Sample beneficials. Consider releasing <i>Trichogramma</i> into sorghum. Keep track of the BDI and predator / beneficial to pest ratio. Food sprays may be considered. Manage lucerne appropriately.	Sample for beneficials. Encourage beneficials to reduce late season resistant pests through food sprays and consider low impact insecticide options.
4. Prevent the development of resistance	Pupae bust to control overwintering <i>Helicoverpa</i> and mites as soon as possible after harvest. Plant spring trap crop. Attend annual resistance management meeting. Reduce the availability of aphid and whitefly hosts over the winter.	Consider Bollgard II® refuge options. Consider choice of at-planting insecticides or seed treatments and implications for later aphid sprays.	Use pest and damage thresholds. Follow the IRMS strategy for the region. Encourage beneficials to help reduce resistant pests. Follow Bollgard II® resistance management plan.	Use pest and damage thresholds. Follow the IRMS strategy for the region. Encourage beneficials to help reduce resistant pests. Follow Bollgard II® resistance management plan.	Use pest and damage thresholds. Follow the IRMS strategy for the region. Encourage beneficials to help reduce resistant pests. Follow Bollgard II® resistance management plan.
5. Manage crop and weed hosts	Keep farm weed free over winter. Control cotton re-growth.	Carefully consider summer rotation crops (type and location). Keep farm weed free.	Keep farm weed free.	Keep farm weed free.	Consider winter rotation crops (type, location and the potential to host pests or diseases). Keep farm weed free.
6. Using trap crops effectively	Plant spring trap crop. Consider flowering date to time planting.	Consider summer trap crop. Cultivate all chickpea trap crops by the 30th September.	Consider last generation trap crop.	Monitor <i>Helicoverpa</i> populations in summer trap crop, control if necessary.	Destroy <i>Helicoverpa</i> eggs and larvae in last generation trap crop using biological sprays. Pupae bust last generation trap crop.
7. Support IPM through communication and training	Consider becoming involved in an IPM or AWM group. Attend regional training and information seminars. Consider doing the IPM short course.	Communicate with neighbours and applicators to discuss spray management plans. Attend training courses i.e. Cotton-LOGIC.	Meet regularly with your neighbours and consultant to discuss IPM strategies and attend local field days.	Meet regularly with your neighbours and consultant to discuss IPM strategies and attend local field days.	Meet regularly with your neighbours and consultant to discuss IPM strategies and attend local field days.



3. IPM strategies - 'how to do it'

3.1 Objective 1 - Growing a healthy crop

3.1.1 Introduction

IPM can help reduce crop losses by pests, but cannot directly improve the yield potential and fibre quality of cotton. These are primarily influenced by crop agronomy and variety choice.

Crop management can affect IPM. For instance, poor fertiliser or irrigation management can affect crop maturity, increasing the length of time that the crop requires protection from pests, which can increase insecticide resistance selection. Excessive fertilisation or irrigation can also produce crops that may be more attractive to pests which may require protection. Thus appropriate water and nutrient management are essential in producing a healthy crop and in supporting an effective IPM system.

This objective covers the key issues for good crop agronomy and highlights how they interact with IPM.

3.1.2 Field selection

Selecting which fields to plant to cotton can affect the success of IPM on the farm. Considerations include proximity to sensitive areas such as rivers, watercourses, stock routes, pastures, domestic dwellings, workshops, neighbours and the prevailing wind direction. As Bollgard II® varieties have a reduced requirement for insecticides to control *Helicoverpa*, they may be appropriate for fields near sensitive areas. Although to help achieve maximum returns, Bollgard II® should also be planted in fields that can be watered effectively and have a history of reliable production. Shorter season varieties may also be considered for sensitive areas as the shorter growing period reduces the time the crop needs to be protected from pest damage.

If a grower is experimenting with IPM tools and strategies on part of the farm, then these 'IPM' fields should be situated so as to reduce the risk of drift contamination from sprays applied to the rest of the farm or to fields on neighbouring farms. Other considerations include the proximity of these IPM fields to other crops or orchards which can potentially act as a source for secondary pests such as mites, aphids or whiteflies. Growers could consider keeping pest resistant varieties together, such as Bollgard II® or okra leaf types, rather than intermingling them with other varieties. This will help reduce the chance of sprays applied to one type of cotton, i.e. conventional, disrupting IPM on other fields that do not need spraying at that time, i.e. Bollgard II®.



A Bollgard II® cotton crop growing in a sensitive area close to a river.

3.1.3 Do I pre-irrigate or water-up cotton?

Growing a healthy cotton crop optimises both its yield potential and capacity to compensate for pest damage. In irrigated cotton, a healthy crop begins with good field preparation, soil moisture and plant establishment.

Unless rainfall provides adequate soil moisture either before planting or directly after planting, pre-irrigating or watering-up the field will be necessary. There are advantages and disadvantages with both methods.

Pre-irrigation before planting

The advantages of pre-irrigating are:

- less cracks in the seed bed at planting for optimum establishment
- the encouragement of weed emergence prior to planting to allow effective application of a pre-plant knockdown herbicide
- warmer soil conditions at planting, as the soil has time to warm up between the irrigation and planting. This may help speed germination and early growth and reduce losses to seedling diseases.

The disadvantage is that hot conditions may cause the soil to dry quickly so that the top of the hills must be knocked off to expose moist soil for planting. This reduces bed height which increases the risk of seedling disease and waterlogging. In some cases the grower may need to 'flush' the field with water after planting to ensure even germination and avoid a 'gappy' plant stand.

Watering-up after planting

Watering-up allows the grower to plant in a more timely manner, especially when planting into standing wheat stubble.

However, watering-up often reduces soil temperatures which can slow seedling vigour and increase the risk of seedling disease. Watering-up also encourages weeds to germinate at the same time as the cotton, and unless the variety is herbicide tolerant, a post-emergent knockdown herbicide is not an option.

3.1.4 Seed bed preparation

A tactic often mentioned by cotton growers in achieving an early crop is a good seed bed, typified by friable, non-cloddy soil and firm, high, well shaped beds. This helps achieve vigorous healthy early growth resulting in plants that are able to tolerate seedling disease better, achieve high yields and early crop maturity. High beds also reduce the risk of waterlogging by encouraging good drainage. Planting cotton into standing stubble (wheat, sorghum) may offer some benefit in terms of soil condition, insect management and water infiltration. However, there are management issues associated with standing stubble, such as an increased risk of waterlogging from heavy rainfall, or rainfall following an irrigation and the potential to reduce soil temperatures. Some of these risks can be reduced by careful management, see section 3.1.8 '*Planting into standing wheat stubble*'. Adopting a system of planting cotton into standing wheat stubble requires a significant change in management in terms of planting regime, fertiliser application, equipment for sowing and irrigation. For more information see the publication '*Planting Cotton in Standing Wheat Stubble*', available from the TRC or Australian Cotton CRC website.

3.1.5 Selecting a cotton variety

The cotton variety planted should be matched to the region and likely pests and diseases (see seed company variety guides or websites). Planting a variety with a long growing period and a high yield potential in a cooler, shorter season region is likely to create problems with late maturity, poor fibre quality, prolonged protection and difficulty with defoliation. This issue is important if wet weather delays ground preparation.

Okra leaf varieties have a degree of resistance to both *Helicoverpa* spp., spider mite and silverleaf whitefly, which potentially reduces sprays for each pest by about 1 per season. Penetration of insecticides into the crop canopy is also better with okra leaf cultivars, which can contribute to better control.

Disease tolerance is also an important consideration. Selection of tolerant



Watering-up a cotton field after planting allows more flexibility with the timing of planting, but may decrease soil temperature, promoting seedling disease.



Okra cotton varieties (left) are more tolerant of *Helicoverpa*, mite and silverleaf whitefly.



Coordinating planting in a region to a specified planting window avoids a wide spread of crop maturation and prolonged insecticide use. This helps manage insecticide resistance and support IPM.



Checking the establishment of an Ultra Narrow Row experiment.

varieties is advisable in fields with a history of verticillium or fusarium wilt. Planting of a Cotton Bunchy Top (CBT) resistant variety, in areas where there is a risk of a high aphid population e.g. following a wet winter, or in a field with poor weed control. This will reduce the risk of the crop being infected with CBT, thereby reducing the temptation to control aphids at very low densities to prevent the spread of CBT and the associated risk of developing insecticide resistant aphids.

3.1.6 Planting window

Use of a specified period for planting, known as a planting window, reduces the spread of crop maturation in a region at the end of the season, which helps avoid very late crops that require pest management for a prolonged period. Such late crops increase the risk of selecting for insecticide resistance. In some regions, e.g. Central Queensland, planting windows are used with Bollgard II® management to help with their insecticide resistance management strategy.

In each cotton region there is a period when the soil temperatures become suitable for cotton germination. Planting at this time usually maximises plant establishment and avoids the risk of frost. Early soil preparation to optimise soil structure and seedbed tilth will facilitate early planting. Very early planting (mid September) in cool districts increases the risk of cold shock (min temperature <11°C), which slows early growth and reduces tolerance to herbicide damage, seedling diseases and early pest damage, especially from thrips. Late planted cotton runs the risk of declining yield potential, is more susceptible to pests such as whitefly, and late season infestations of *H. armigera* which are difficult and expensive to control because many are resistant to most of the chemicals used against them. A planting window that reduces the time insects are exposed to chemicals, also reduces the number of generations that are exposed, thereby reducing selection for resistance and the likelihood of overwintering pupae developing.

Soil temperature is used to assist in planting decisions. A minimum soil temperature of 17°C at 7.00 am at planting depth for 3 consecutive days before planting is recommended. This ensures rapid germination and good seedling growth. Monitor weather forecasts for impending cold fronts.

Planting windows are critical to the success of area wide management strategies for monocultures such as cotton. Planting windows limit the time period during which cotton can be planted. This mainly seeks to reduce the spread of maturity dates, generally avoiding late cotton crops. By limiting planting dates, there is the opportunity to reduce exposure of cotton to pests and hence the need for pest control. This is particularly an issue for Bollgard II® cotton, where prolonged exposure by having late crops, will expose the technology to more generations of *Helicoverpa* spp.

Plant establishment

It is desirable in irrigated cotton, in most cotton regions, to have 8-12 plants per metre of row, distributed along the row as uniformly as possible. However, plant densities as low as 4 plants per metre will produce near maximum yield as long as the stand is uniform. Where factors like soil type may restrict the plant size, the optimum plant density should be at the higher end of the range, i.e. 12 plants per metre.

For dryland cotton systems the plant density should be about ½ that of irrigated cotton along the row, i.e. 6-8 plants per metre of row for a skip row or solid planting configuration. For Ultra Narrow Row (UNR) intended for finger stripper harvesters, the plant density should be about 25-35 plants per square metre, or for 15 inch cotton intended for spindle harvesting, about 20-25 plants per square metre.

3.1.7 Optimising earliness

Although managing a crop for earliness is a good strategy, it does not always maximise yield. Figure 2 presents two years of data from two

different sowing times and a range of cultivars with varying maturity potentials. This shows that decreasing the days from sowing to maturity also decreases lint yield. Therefore both the advantages and potential disadvantages of growing an early crop should be weighed up.

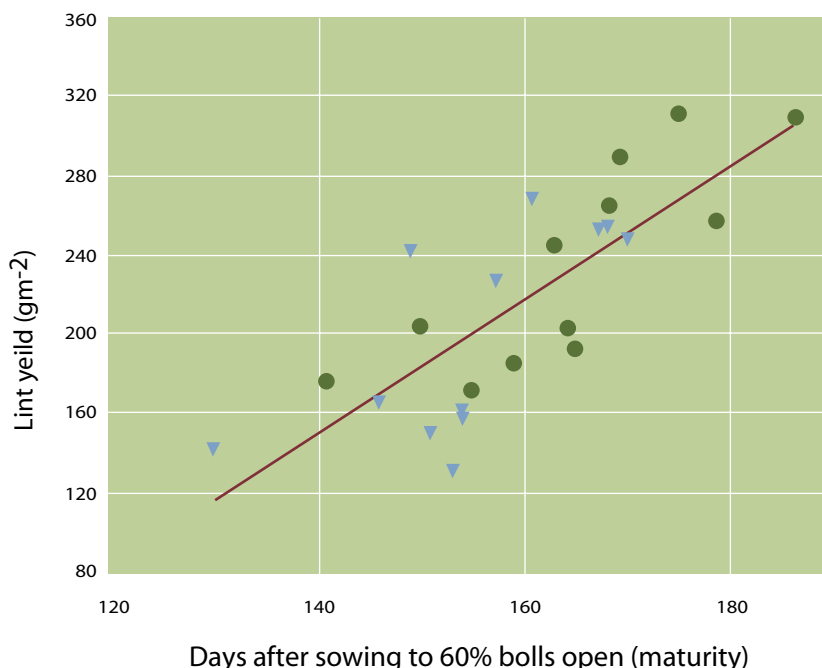


Figure 2.

Relationship between yield and crop maturity for a range of cultivars, years and sowing dates at Narrabri. For each week that the crop matures earlier, yield decreased by about 1 bale / ha. Similar relationships have been found in a number of studies. (Milroy *et al.* 2002. Is earliness really next to Godliness? Proceedings 11th Australian Cotton Conference 13th - 15th August 2002, Australian Cotton Growers Research Association Inc.)

For Bollgard II® crops with the potential to produce a higher boll load, research has shown that later sowing times have not effected the time taken to reach crop maturity. Sowing later has a number of potential benefits:

- 1 Better germination and seeding survival with warmer soil temperatures. With increased seedling vigour, the plants have a better chance of surviving seedling diseases. Increased seedling survival improves plant stand.
- 2 Avoids high temperatures during fibre development.
- 3 Can achieve a bigger plant before fruit development to help maximise yields.

For more information on how to manage a crop for early maturity, refer to the 'Integrated Pest Management Booklet' - Objective 1. 'Manage the crop for early maturity' in the 'Australian Cotton Industry Best Management Practices Manual'.

3.1.8 Planting into standing wheat stubble

Planting cotton into wheat stubble can help minimise the off-farm movement of pollutants such as fertilisers and insecticides. The wheat stubble slows the movement of water off a field, thereby reducing the movement of sediments that may contain pollutants. A number of other benefits such as improved water infiltration, increased organic matter, improved soil condition at planting and during seedling growth and reduced early season *Helicoverpa* pressure have been shown in some studies.

Planting cotton into standing wheat stubble can offer benefits for an IPM system but brings new challenges to the farming system. Wheat stubble can slow the movement of water across the field and increase water infiltration which increases the chance of excess deep drainage and waterlogging. Consequently, adjusting the irrigation technique may be necessary. Additionally, it is possible to reduce the risk of waterlogging by removing stubble from the furrows, except for the last 20 metres near the tail drain. This improves water flow, reduces the risk of waterlogging, but the stubble in the furrow catches sediment before it can be carried off the field. (Hulugalle *et al.* Aust. Cottongrower magazine Oct-Nov 2004 pg. 58-62.)



Planting cotton into wheat stubble.

Crop nutrition may also need modifying to avoid nitrogen problems. These changes will vary depending on factors such as soil type and climate.

3.1.9 Seed treatments, granular insecticides and in-furrow insecticides



Granular insecticides applied at planting do not effect beneficial populations such as the bigeyed bug.



Large false wireworm larvae attack seedling cotton and if severe can cause 'gappy' plant stands.



Thrips are pests but also predators of mite eggs.

The main pests targeted by seed treatments, granular insecticides and in-furrow insecticides are wireworms (false and true wireworm), early thrips, mirids, mites and black field earwigs. Sprays applied to the seed furrow at planting can provide good control of these pests for 4-6 weeks after emergence. Seed treatments can provide moderate control of thrips for about 2-4 weeks after emergence, and light to moderate control of wireworm infestations. Granular insecticides applied into the seed furrow at planting will provide good protection against wireworms and control of thrips, aphids, mirids and mites for 4-6 weeks after emergence depending upon the product type and rate used. The decision to use any of these products must be made before there is a pest problem, which means their use is 'prophylactic'. This does not mean they are incompatible with IPM, as they are selective against many beneficial groups. They are likely to have less impact on beneficials than a foliar application of an insecticide that targets thrips or mirids (refer to the latest '*Cotton Pest Management Guide*'). Their selectivity is based on the fact that they do not contaminate the surface of foliage but are absorbed by the plants. Since most beneficials do not directly feed on cotton foliage, they are unaffected by the insecticide.

Wireworms attack the top of the root just below the soil surface and can cause a reduction in plant stand. They tend to be worse in fallow fields with high levels of stubble and following dry winters, and less of a problem in back to back cotton where insecticides used against other pests usually reduce the abundance of the adult beetles. The decision to control wireworms should be made only after sampling the soil for the pest. This decision must be made before planting, as wireworms cannot be controlled after plant establishment without destroying the plant population.

Thrips are both a seedling pest and a predator of mites, so they should only be controlled where there is a reasonable expectation of an economic benefit. In cool regions (Upper Namoi, Hillston, parts of the Darling Downs and Macquarie) controlling thrips will give a significant yield benefit in 1 year in 2, while in warmer regions control of thrips will give a significant yield benefit in only 1 year in 10 and thrips will cause little, if any, delay in crop maturity (average about 4 days). Plants may have initial leaf area significantly reduced, but they generally recover from the damage 40-60 days after sowing, without yield loss or delay. Thus use of longer lasting and more expensive control options such as a seed treatment or at-planting insecticide (granular or sprayed) is rarely justified in warm areas, but should be considered in cool areas. Treated seed will provide short term control of thrips, facilitating establishment of the seedling plants and is compatible with IPM. As the effect of the seed treatment diminishes and the crop grows and becomes less sensitive to thrips damage, thrips can re-infest and help control mites by eating their eggs.

Selection of seed treatments may also interact with spider mite management, as thrips are important predators of mites. Some insecticides control thrips well but do not affect mites, thereby increasing the potential risk of mite outbreaks. Some seed treatments or in-furrow insecticides can help to control spider mites on seedling cotton for about 4-6 weeks depending upon the rate which may delay the development of mite populations (refer to the latest '*Cotton Pest Management Guide*').

Often other ground dwelling insects are present in fallow fields, but these have little or no impact on cotton seeds or seedlings. For example, millipedes are usually abundant very early in the season, however they prefer to feed on decaying matter rather than fresh plant material. Others such as centipedes and carabid beetles are regarded as beneficials.

Another consideration in the decision to use treated seed or an at-planting insecticide is the risk of the crop being colonised by aphids. Some of these aphids may be carrying the agent for the disease known as Cotton Bunchy Top (CBT). A number of the insecticide options will control aphids, potentially reducing the risk of yield loss from this disease. Growers planting in fields that had CBT the previous season should consider using an at-planting insecticide or seed treatment effective against aphids following the resistance management guidelines found in the '*Cotton Pest Management Guide*'.

3.1.10 Water budgeting and interaction with nitrogen

Irrigation should be based on information from neutron probes or other soil water measuring instruments and / or an irrigation management program such as HydroLOGIC. Irrigation decisions should be based on crop requirements and the recognised soil water deficit for the particular soil. Too much nitrogen fertiliser and water can promote excessive growth and therefore the need for an additional irrigation late in the season. This can cause late season problems with the control of *Helicoverpa* spp., and can undermine the value of 'last generation' trap crops, as the cotton crop will be very attractive to pests.

More information on water management can be found in *WATERpak*, available from the TRC or on the Australian Cotton CRC website.



Using a neutron probe to measure soil moisture.

3.1.11 Nitrogen rate and crop nutrition

The amount of nitrogen available to the crop affects pest management, yield potential and maturity. Too little nitrogen will prevent the crop from achieving its yield potential. Too much nitrogen creates excessive cotton growth toward the end of the season. This makes the crop more attractive to pests, requiring additional inputs of insecticides (and mixes) for control, and application of high rates of growth regulators to retard growth. It also undermines the effectiveness of the 'last generation' trap crop by maintaining the attractiveness of cotton relative to the trap crop (refer to objective 6 '*Using trap crops effectively*').

Excessive nitrogen can also delay crop maturity by 1-2 weeks and make crops harder to defoliate. Nitrogen should be managed on a field by field basis and the soil sampled in winter or early spring to determine the background level of nitrogen. The web-based NutriLOGIC decision support tool can then help determine the optimal nitrogen rate for each field, based on the results of soil tests. Petiole nitrate assessments can be used during early crop growth to determine the need to apply supplementary nitrogen during the season. NutriLOGIC can be found on the Australian Cotton CRC website.



Excessive use of N fertiliser can cause rank growth and delays in maturity.

Most nitrogen is applied to the soil in winter or early spring. The timing and method of nitrogen application should minimise the risk of excessive losses due to denitrification. However, this needs to be balanced against the risk of wet weather preventing timely application. At this time other nutritional needs of the crop should also be considered as indicated by the soil tests.

For Bollgard II® crops with the potential to produce a higher boll load, it is possible that the demand for nitrogen may be earlier and greater. Therefore to ensure that there is adequate nitrogen available when the crop needs it, monitoring soil and plant nitrogen to determine optimum fertiliser rates is essential. However the plants ability to extract nutrients is influenced by the soils physical and chemical status. For example, increasing the fertiliser rate for a Bollgard II® crop grown on a compacted field will not result in increased yields. Therefore selecting the best fields for your Bollgard II® crops will help achieve maximum returns.

Adequate nutrition will ensure healthy growth of plants that are more tolerant of pests and diseases. Further information on cotton nutrition can be

found in *NUTRIpak*, which is available from the TRC or Australian Cotton CRC website.

3.1.12 Growth regulators

Optimal irrigation scheduling and nitrogen rates will generally prevent excessive vegetative growth, apart from during hot growing conditions. Such growth is a problem because it reduces the retention of fruit and can delay harvest. Reduced efficacy of insecticides due to poor penetration of the canopy is also a problem.

Use of growth regulators is recommended if required according to the guidelines published by the cotton seed companies. Appropriate use of growth regulators can help to reduce the likelihood of a rank crop that will not cut-out. Growth regulators are also occasionally used at or near cut-out, to reduce the amount of fresh regrowth and the attractiveness of the crop to pests. This strategy is used to lessen the likelihood of late pest infestations and reduce the number of late season sprays.

See the Cotton Seed Distributors website (www.csd.net.au/) for information on calculating vegetative growth rates to determine crop needs, or the Deltapine website (www.deltapine.com.au/) to view the Pix® response guide.

3.1.13 Final irrigation

3.1.13.1 Objective of the last irrigation

The prime objective of the last irrigation is to ensure that boll maturity is completed without water stress, and at the same time prevent the occurrence of lush vegetative growth in crops late in the season to avoid the crop being attractive to *Helicoverpa* spp.

At the time of the last irrigation all bolls have been set, vegetative growth is limited and the majority of carbohydrates are used to satisfy boll demands. The abscission layer to cause boll shed cannot form once a boll reaches 10-14 days old. This is why boll numbers are not significantly reduced by late water stress. However fibre development can be affected resulting in less secondary thickening of cotton fibres (reflected in micronaire), reducing yield and potentially resulting in immature fibres.

Crops that come under stress prior to defoliation, which normally occurs at 60% of bolls open or 4 Nodes Above Cracked Boll (NACB), can suffer some yield and fibre quality reduction. The level of yield reduction increases the longer the stress occurs.

3.1.13.2 Crop water use

End of season water requirements can be estimated from the date of the last effective flower which is when the Nodes Above White Flower (NAWF) measurement is equal to 4. The last harvestable bolls take 600 to 650 degree days to reach maturity. Therefore for crops to be defoliated towards the end of March, the last effective flower needs to occur in the last week of January. Crop water use needs to be considered for this period. At the time of first open boll, crop water use may be 5-7 mm per day and may decline to around 4 mm per day prior to defoliation.

3.1.13.3 Determining end of season crop water requirements

Factors to consider:

1. Days to defoliation
2. Boll maturity
3. Crop water use
4. Plant available water - ability to extract water below normal refill point
5. Soil moisture objective at defoliation

Days to defoliation (general example - need to generate values for your own district)

- Defoliate when NACB is equal to 4



Optimising timing and amount of water applied will help achieve good yields, increase water use efficiency and avoid late crops which are attractive to pests.

- Takes 42 degree days, around 3 days (up to 4 days in cooler regions) for each new boll to open on each fruiting branch
 - $(\text{Total NACB} - 4) \times 3 = \text{days to defoliation}$
 - Aim to be at or close to refill point at time of defoliation
- Two examples are listed below on final water requirements.

Table 2. Timing of irrigation

If refill deficit for the particular soil is 70 mm

	Crop A	Crop B
Total fruiting branches	13	13
% open bolls	25-30%	Zero
NACB	9	13
Days to defoliation	$(9-4) \times 3$	$(13-4) \times 3$
(NACB = 4)	15	27
Estimated daily water use until defoliation	5 mm / day	5.5 mm / day
Total Water Requirement	75 mm	148 mm

Crop A Irrigate now?

- This will depend on the capacity of the crop to extract moisture below its normal refill point. If the crop can extract moisture to 90 mm, at the end of the season, and there is 35 mm (half a profile) of available water still in the profile, irrigation may not be necessary. However if the crop can not extract below 70 mm, an irrigation may be recommended (even if there is 35 mm left in the profile).

Crop B Requires close to two full irrigations.

- Rainfall needs to be considered in such decisions. (don't forget to apply a rainfall efficiency of 40-50%)

Impact of late water stress

- *One irrigation short*
If the crop is one irrigation short (i.e. reaches refill point at 20% of bolls open), boll size will generally be reduced rather than resulting in a significant reduction in boll numbers. This will reduce yield and can also affect fibre quality (reduced micronaire, little effect on length).
- *Two irrigations short*
Boll numbers will be reduced. Provided the crop does not move into rapid stress, boll size may actually be increased due to the shedding of younger bolls (< 10-14 days old). However, the loss of these younger bolls is a loss of potential yield. Fibre micronaire may be increased in the remaining larger bolls, while length and micronaire may be reduced on those younger bolls that do survive.

Significant yield reductions are likely to occur because the boll number is reduced. In large vegetative crops that come under stress prior to boll opening, both boll size and number can be reduced, with significant reduction in yield and fibre quality.

OZCOT simulation - dry harvest years

OZCOT is the crop development model developed by the CSIRO in Narrabri. Using historical weather data from past years, a range of yield reductions can be predicted from late water stress as shown in Table 3.

Table 3. Yield predictions from late water stress

Stage at which stress occurs	Average yield (b / ha)	Yield reduction (b / ha)	Range (b / ha)
nil - full irrigation (average)	8.37		
20% of bolls open (one irrigation short)	7.77	0.60	0.2 - 1.1 (2% - 13%)
6-8 days prior to first open boll	7.11	1.2	0.8 - 1.9 (10% - 23%)

In terms of value per megalitre, if the last irrigation takes 70 mm of water (including losses) and the average benefit of the irrigation is 0.60 bales per ha, then the value of the extra water is 0.86 bales per megalitre.



The 'HydroLOGIC' decision support package allows growers to schedule irrigation, and ask 'what if' questions regarding irrigation options and yield outcomes.

HydroLOGIC - irrigation management software

HydroLOGIC helps cotton farmers manage their irrigations better and improve water use.

The primary aim of HydroLOGIC is to assist in the effective and timely application of irrigations for furrow irrigated cotton crops. It is also able to provide information to help growers assess the consequences of different irrigation strategies on crop growth, yield and water use.

Based on information such as the soil moisture deficit, fruit load, leaf area and historical climate data, HydroLOGIC uses the OZCOT model to simulate the most likely outcome, and report on yield and overall water use with different management strategies.

HydroLOGIC is provided free to Australian cotton growers and consultants. For more information or to receive a copy contact your local cotton industry development officer, the TRC or visit the Australian Cotton CRC website.

3.1.14 Defoliation

You can assess maturity by counting the nodes above the first position cracked cotton boll (NACB).

Defoliate promptly when the crop is mature. The safe timing of defoliation is when the youngest boll expected to reach harvest is physiologically mature. This usually occurs when 60-65% of bolls are open. The other method of assessing physiological maturity is when there are 3-4 nodes of first position bolls above the highest cracked first position boll (last harvestable boll), known as Nodes Above Cracked Boll (NACB).

Crops should only be defoliated earlier than the above recommendations if you are confident that the upper bolls are sufficiently mature, otherwise a reduction in yield or fibre quality could occur. The maturity of bolls can be assessed with care by cutting bolls and checking the development of the seeds. Cotton bolls are mature when the fibre is well developed, the seeds are firm and the seed coats are turning brown in colour.

The timing of defoliation can be an important IPM tool, as late *Helicoverpa* problems can sometimes be overcome by a successful defoliation. This can minimise the need for late season insecticides.



3.2 Objective 2 - Keeping track of insects and damage

3.2.1 Introduction

Sampling pests, beneficial insects and damage provides the basis on which pest management decisions are made. Objective, accurate sampling is essential for IPM to make optimal use of management tactics. Details are provided here for sampling techniques appropriate for both cotton pests and beneficials (to calculate the predator / beneficial to pest ratio) and for pest damage.

Crops should be checked frequently for pests and beneficials and for damage or fruit retention. Frequent checking allows pest populations and damage to be detected early. This gives flexibility to the system, allowing for the action of beneficials and natural mortality (due to hot weather or rain for instance) to occur between checks, without the pest population developing to a stage where control is impractical or too expensive. This makes the need to spray as soon as a pest exceeds threshold less urgent. In contrast, infrequent checking tends to encourage use of insecticides as an insurance to control the pests before the next check in 4 or 5 days time.

This objective covers insect sampling techniques, specific information on pests and beneficials, pest thresholds, monitoring plant damage and pest management decision making.



Visual sampling is clearly superior to d-vacing or beat sheeting for monitoring all *Helicoverpa* spp. life stages.

3.2.2 Sampling beneficial insects and spiders

D-vac, beat sheet or sweep net sampling can be used as an alternative to visual sampling for beneficial insects and spiders. If the grower, consultant or agronomist decides to use these methods to assess beneficials, then sampling should be done on the same day as visual checks are made for pests. Beat sheets are more effective for finding beneficials than the visual or d-vac methods. Refer to the section '*Sampling techniques*' in this objective for more detailed information on the beat sheet technique.

For a good estimation of beneficial numbers, the best time to sample is before 12 noon or late in the afternoon. As the temperature rises throughout the day, many pests and beneficials tend to move down the plant and seek shelter in the lower plant structures and even in the soil making them difficult to find and count.

Sampling of lucerne strips or other refugia crops to assess predator abundance should use a similar method but d-vac sampling is the most appropriate and fastest method to assess beneficial insect populations in lucerne. Select areas for sampling at random from 2 different sites in the field. After sampling, the contents of the d-vac should be carefully transferred to a clear plastic bag to facilitate predator counting. Table 4 lists the main predatory insects and spiders found in Australian cotton crops.

It is important to consider natural levels of *Helicoverpa* egg parasitism

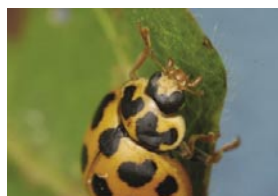
caused by parasitoids such as *Trichogramma*. Consequently, growers should make every effort to consider parasitism levels when making spray decisions. This is best achieved by assessing the levels of pest parasitism as explained in the section ‘*Sampling and determination of Trichogramma parasitism*’ in objective 3.

Some insect species are both predators and pests. For example, thrips are seedling pests of cotton but also eat eggs of mites, contributing to their control. Similarly, apple dimpling bugs are also predators of mites and *Helicoverpa* spp. as well as being a pest if present in high numbers.

For further information about beneficial insects found in Australian cotton IPM systems, visit ‘*The Pest and Beneficial Guide*’ on the Australian Cotton CRC website.

Table 4. Examples of predatory insects identified from cotton farms

Order	Family	Species	Common Name	Group
Coleoptera	Coccinellidae	<i>Coccinella transversali</i>	Transverse ladybird	Predatory beetles
		<i>Domus notescens</i> <i>Harmonia Octomaculata</i> <i>Hippodamia Variegata</i> <i>Dicranolaius bellulus</i>	Two-spotted ladybird Three banded ladybird White collared ladybird Red & Blue beetle	
Hemiptera	Nabidae Lygaeidae Pentatomidae Reduviidae	<i>Nabis kinbergii</i> <i>Geocoris lubra</i> <i>Cermatulus nasalis</i> <i>Ochelia schellenbergii</i> <i>Coranus trabeatus</i>	Damsel bug Bigeyed bug Glossy shield bug Predatory shield bug Assassin bug	Predatory bugs
Neuroptera	Chrysopidae Hemerobiidae	<i>Chrysopa</i> spp. <i>Micromus tasmaniae</i>	Green lacewing Brown lacewing	Predatory lacewings
Araneida	Lycosidae Oxyopidae Salticidae Arenidae	<i>Lycosa</i> spp. <i>Oxyopes</i> spp. <i>Salticidae</i> spp. <i>Araneus</i> spp.	Wolf spider Lynx spider Jumping spider Orbweaver	Spiders



Three banded ladybird



Transverse ladybird



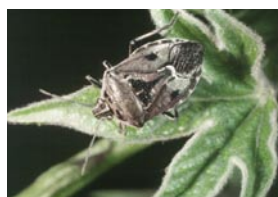
Ladybird larva



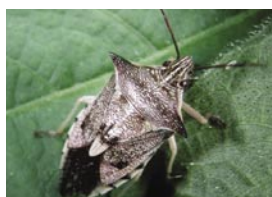
Red & Blue beetle



Bigeyed bug



Glossy shield bug



Predatory shield bug



Damsel bug



Assassin bug



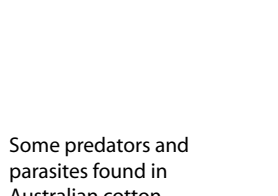
Hoverfly



Green lacewing adult



Lynx spider



Some predators and parasites found in Australian cotton crops.



Orbweaver spider



Trichogramma wasp



Orange caterpillar parasite

3.2.3 Sampling techniques

Insect numbers should be recorded either as numbers per metre or as a percentage of plants infested to easily compare numbers with the appropriate threshold and to allow a predator / beneficial to pest ratio to be determined. The CottonLOGIC decision support tool supports a number of sampling techniques and automatically converts pest abundance to a standard numbers per metre, compares this with the control threshold, and also calculates the predator to pest ratio (refer to ‘*Computerised decision support for pest management*’ in this objective).

Recent studies have shown that *Helicoverpa* spp., whiteflies, mites, aphids, thrips and apple dimpling bug nymphs are best sampled visually throughout the entire season, while the beat sheets are superior for the majority of other insects and spiders. *Trichogramma* however requires a specific sampling technique which is detailed in the section ‘*Sampling and determination of Trichogramma parasitism*’ in objective 3.

Visual sampling: Whole plants should be sampled. This involves checking the entire plant, including squares and around bolls. This will ensure that pests or predators, which are present lower in the canopy, are recorded. Check at least 30 plants or 3 separate metres of cotton per 50 ha. Larger sample sizes give more accurate estimates of insect numbers, leading to more reliable pest management decisions. Fields are rarely uniform in crop growth and attractiveness to insects. Lush areas, such as near the head ditch, are more attractive to insects. By being aware of such areas and their size you will be more able to determine how many sample entry points are required in a variable crop.

Beat sheet sampling: A standard beat sheet is a sheet of yellow canvas 1.5 m x 2 m in size, placed in the furrow and extended up and over the adjacent row of cotton. A metre stick is used to beat the plants 10 times against the beat sheet, moving from the base to the tops of the plants. Insects are dislodged from the plants onto the canvas and are quickly recorded. Sample at least 3 m per 50 ha, although larger sample sizes give more accurate estimates of insect numbers

Beat sheet sampling in a field that has been recently irrigated can become a very dirty task as the mud sticks to the sheet. This technique can also be difficult to use when there has been a heavy dew, as once the plants have been pushed onto the sheet, many insects will stick to the water droplets on the plant or on the sheet and are difficult to see.

Beat sheet sampling can detect about twice the number of total predators than visual sampling after the crop reaches 9 to 10 nodes. These differences must be kept in mind when calculating the predator / beneficial to pest ratio, as this ratio is currently based on visual counts. This information relating to the beat sheet method is preliminary, and is subject to modification following further studies.

Beat sheets are also effective for sampling mirids, and are more efficient than visual samples. To convert mirid numbers from beat sheet to a visual equivalent, divide the beat sheet count by 3 after the crop reaches 9-10 nodes. Before the 9-10 node stage the beat sheet and visual counts are similar.

If you are finding much larger differences in the number of insects detected on a beat sheet to your visual counts, it would be advisable to develop your own conversion by dividing the beat sheet count by the visual count. These counts should be carried out at the same location (about 5 m from each other) at a similar time on the same day. For advice in developing your own conversion contact the TRC.

D-Vac sampling: For d-vac sampling, a standard, petrol powered garden blower / vacuum machine, of the type made by Homelite or Stihl is used. The machine should have a suction tube of 120 mm in diameter. A gauze bag should be inserted into the suction tube to collect insects sucked from



Counting insects on a ‘beat sheet’.



D-vac sampling is reliable for estimating beneficial numbers when plants are small.

the plants. During d-vac sampling the tube of the d-vac should be drawn from the top to the base of the plants. Generally 20 m of row is sampled, which should be divided into 4 random samples of 5 m for a more accurate sample.

D-vac sampling is reliable for estimating beneficial numbers when plants are small. After the plants have about 9 nodes the reliability of the d-vac sampling method declines.

Templates for the gauze bags can be obtained by contacting the Australian Cotton Research Institute 02 6799 1500.



Using a sweep net to sample insects in a pigeon pea trap crop.

Sweep net sampling: Sweep net sampling is good for many beneficials and mirids in the upper canopy. As shown in the image on the left, a sweep net is a large cloth net (approximately 60 cm deep) attached to a round aluminium frame about 40 cm in diameter with a handle (1 m in length). Each sample can consist of between 20 and 50 sweeps across a single row of cotton. Walk briskly and sweep the net in front so that the bottom of the sweep strikes the canopy about 25-30 cm from the top. Keep the net moving fast enough so that flighty insects such as mirid adults can not fly out. For pest managers using the sweep net to supplement their visual counts, two samples of 20 sweeps on the way to do their visual sample and walking out of the crop has been suggested. After each sample, the net is then carefully assessed for the presence and / or number of particular insects captured. A larger sample size will give a more accurate estimate of insect numbers.

Studies are being conducted to validate the effectiveness of the sweep net technique and to calibrate the number of insects detected to a visual equivalent.

Frequency of sampling

Pests and beneficials should be sampled at least every 2-3 days. Re-commence checking within 3 days of a spray, but do not re-enter crops within the prescribed re-entry period for the insecticide applied (refer to the chemical's label or safety data sheet). In the hotter part of the season, it is essential to check regularly as insect pressure is usually greater and development times for insects faster. *Helicoverpa* development can be estimated using CottonLOGIC, which can predict the development of a population for 3 days following a sample. CottonLOGIC uses a model of *Helicoverpa* growth, which considers the region, time of the season, temperature and natural field mortality.

The edges of fields are often quite different to the rest of the field, so leave a buffer zone around the edges of the field i.e. do not sample within 50 m of the edge unless required for specific aphid or mite monitoring. To allow for variation in the field, try to check in different parts of the field. You should try to rotate sampling areas within the field on different occasions. Try to check across the normal direction of spraying.

3.2.4 Sampling notes for individual pests

Helicoverpa spp.

As a major pest, it is vital that *Helicoverpa* sampling and recording is done accurately in both conventional and Bollgard II® crops. Record numbers of egg and each larval category per m. Below is a guide to the characteristics of the eggs and the sizes of the larvae:

White eggs	Egg, pearly white in colour
Brown eggs	Egg, off white to brown
V.S. larvae	Very small larvae, 0.0 - 3.0 mm
S. larvae	Small larvae, 3.0 - 7.0 mm
M. larvae	Medium larvae, 7.0 - 20.0 mm
L. larvae	Large larvae, greater than 20.0 mm

Helicoverpa is most accurately sampled throughout the entire season using

H. armigera (right) showing a saddle of darker pigments on the forth segment from the head and white hairs on the first segment compared to the black hairs on *H. punctigera* (left).



visual methods. CottonLOGIC supports all visual methods including numbers / metre, numbers / plant and presence / absence. Presence / absence is a binomial sampling technique which records the presence or absence of *Helicoverpa* on plant terminals rather than absolute numbers on whole plants.

Identifying the *Helicoverpa* species

Correct identification of *Helicoverpa punctigera* and *H. armigera* is important for the effective use of many insecticides. This is because *H. armigera* is resistant to a range of insecticides, especially the pyrethroids, endosulfan and carbamates, whereas *H. punctigera* is not. The two species can be separated visually during some stages of their life cycle, e.g. medium and large larvae, pupae and adults. It is not possible to visually differentiate *Helicoverpa* eggs or early larval stages. Medium larvae can be identified by the presence of a 'saddle' of darker pigments on the fourth segment back from the head. Large larvae can be distinguished by the colour of the hairs on the first segment behind the head, white for *H. armigera* and black for *H. punctigera*. Pupae can be separated using two small 'tail' spines which are apart and slightly smaller in *H. armigera* and close together and longer in *H. punctigera*. Adults are identified using their hind wings, with *H. armigera* having a small light patch in the dark section of the hind wing. The dark section on *H. punctigera* is uniform.



Adult moths of *H. armigera* (right) have a pale patch on the hind wing, which is not present in *H. punctigera* (left)

If you experience any difficulties in correctly identifying the species or would like some specific information on which species may be dominant for a particular time of the season, contact your local industry development officer.

Tipworms (*Crociosema plebejana*)

Sample for tipworms up until first flower. Record the number of tipworm eggs (white and red), small larvae (< 3 mm) and large larvae (> 3 mm) per m. As larvae such as tipworm or *Helicoverpa* tend to burrow in the terminals, bolls and squares and may not be found using a beat sheet or d-vac, the visual sampling methods are the most accurate.

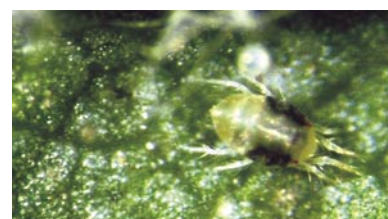


Large tipworm larva. These can bore into the terminal of young cotton plants.

Two-spotted spider mites (*Tetranychus urticae*)

A visual presence / absence method is used to sample mites to account for their patchy distribution within a crop. There is a strong relationship between the % of leaves infested with mites and the number of mites per leaf. Using this sampling protocol, leaves are sampled and rated as infested or not infested with mites. This increases the likelihood of finding mites if they are present and is crucial in their effective management. The sampling protocol is given below:

- (a) Walk about 40 m into the field (early in the season it is advisable to sample near the field edges to see if significant influxes of mites have occurred),
- (b) Take a leaf from the first plant on the right or left. The leaf should be from the third, fourth or fifth main stem node below the terminal. If the plant has less than 3 leaves sample the oldest leaf. Until the plant has about 5 true leaves, it may be easier to pull out whole plants.
- (c) Walk 5 steps and take a leaf from the next plant, on the opposite side to the previous one, and so on until you have 50 leaves,
- (d) As the leaves are collected or after all the leaves have been collected score each leaf by turning it over and looking at the underside, firstly near the stalk or petiole, then scan the rest of the leaf. If mites of any stage (eggs or motiles) are present score the leaf as infested. A hand lens may be needed to see mite eggs as they cannot be easily seen with the naked eye.
- (e) Repeat this simple procedure at several widely separated places in the field to allow for differences in mite abundance within the field. Depending on the size of the field, between 4 to 6 sites are needed to obtain a good estimate of mite abundance.



Two spotted mite with egg. Mites will not effect lint yield after 20% of bolls are open.

Sample seedling cotton (up to 6-8 true leaves) regularly to determine the

level of infestation using the standard presence / absence technique described above. If more than 5% of plants are infested it is advisable to count the numbers of mites on plants, and to score the mite damage level (i.e. estimate the % of the plant's total leaf area that is damaged by mites). Continue to monitor mite numbers, damage levels and infestation levels at least weekly, or more frequently if infestation levels are high (> 30% of plants infested). If the level of infestation, damage level or mite number per plant declines then control is unnecessary and monitoring should continue. If mite numbers per plant do not decline after about 6 weeks, if the damage levels exceed an average of 20% of plant leaf area or if infestation levels increase then predators are not abundant enough to control mites and a miticide should be applied. After about 6-8 true leaves, specific mite counts and damage scoring can cease but continue to use the standard presence / absence sampling method until 20% of bolls are opened, after which mites will not affect yield.

Green mirids (Creontiades dilutus)

Record the total number of adult and nymph mirids per metre using either whole plant counts, a sweep net, a beat sheet or a d-vac. On young cotton these techniques all give comparable estimates of mirid abundance. Recent studies have shown that the increase in the volume and surface area within the crop canopy as the season progresses decreases the efficiency of whole plant visual samples, making sweep nets or beat sheets the preferred techniques to monitor green mirid abundance. After the crop reaches the 9 to 10 node stage, there are three times the mirids found on the beat sheet compared with visual counts. It is essential to monitor levels of fruit retention to consider mirid damage when making pest management decisions.



A small boll with GVB damage.

Green vegetable bugs (GVB) (Nezara viridula)

During the period from flowering to 1 open boll per metre, cotton becomes highly susceptible to mirid and GVB damage. As the type of boll damage is similar for both pests, it is essential to find out which pest is present. Crops should be inspected regularly using a beat sheet to assess populations, and bolls checked for damage. In the field, the distribution of GVBs is generally patchy and therefore thorough inspections throughout the crop are necessary. GVBs are most visible early to mid morning making checking easier at this time. Recent studies have shown that at early squaring, both visual and beat sheet sampling methods are equally effective, however at late stages (after flowering), the beat sheet method is three times as efficient. Damage to small bolls (14 day old) can be assessed by cutting the boll or squashing the boll to check for the presence of warty growths or brown staining of the lint.

Apple dimpling bugs (ADB) (Campylomma liebknehti)

Record the numbers of ADBs per metre. Note that the ADB is both a pest and a predator. It is much less damaging than the green mirid and only warrants control if numbers are extremely high and fruit retention is low (< 50% of first position fruit). ADBs are best counted visually on whole plants, as the beat sheets are usually yellow in colour which makes counting the yellow ADB nymphs very difficult. Early season d-vac sampling is also effective in detecting ADB populations.

Sticky aphid honeydew on a cotton leaf (left) compared to a clean leaf.



Aphids and honeydew

Aphids are sampled on a presence / absence basis by scoring the number of plants infested with aphid colonies. A colony is defined as 4 or more aphids within 2 cm. Identification of the aphid species present is critical as they differ in resistance to insecticides. Cotton aphid (*Aphis gossypii*) is most prevalent but the green peach aphid (*Myzus persicae*) is also sometimes a problem. Distinguishing features of these two species can be found in 'The Cotton Pest and Beneficial Guide'. Aphids are most abundant on the edges of fields so these areas should be checked, especially after the bolls have started to open. Aphids secrete a substance known as honeydew. It gives the plant a sticky feel and can be seen as shiny honey-like spatters on the

leaves. This is a problem because cotton fibre contaminated with honeydew suffers a price penalty. Any trace of honeydew is over threshold once bolls are open and the aphids should be controlled to prevent lint contamination. Often the infestation of aphids will only be on the edge of the field so a border spray may be enough for control.

Silverleaf whiteflies

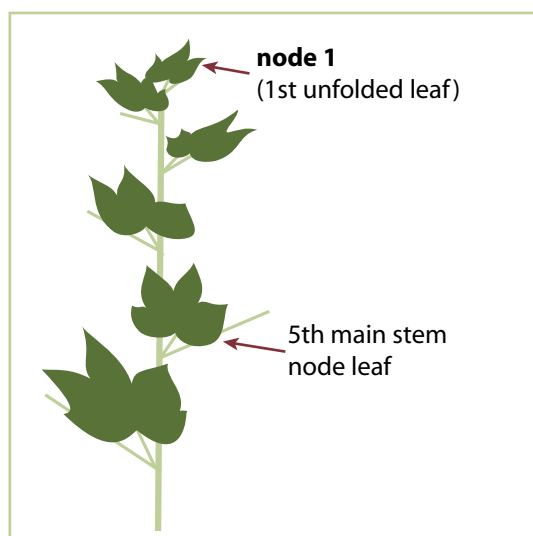
Effective sampling to assess the abundance of both adults and nymphs is critical to identifying which species of whitefly is present before management strategies are implemented. Information on the distinguishing features of the two whitefly species, greenhouse whitefly (*Trialeurodes vaporariorum*) and *Bemisia tabaci* can be found in the cotton pest and beneficial guide. However, there are two biotypes of *Bemisia tabaci*, the eastern Australian native and the introduced B-Biotype which is also known as the silverleaf whitefly. These two biotypes cannot be distinguished visually. Biochemical tests are required to separate the species.

Contact your local industry development officer for information on where to send samples of whitefly for identification.

The silverleaf whitefly is a bigger threat than the other species as it has a wider host range, a higher reproductive rate, develops insecticide resistance rapidly and is an effective carrier of viruses. It also secretes large quantities of sticky honeydew that interfere with photosynthesis and causes problems with cotton fibre processing. Whitefly honeydew is considered worse than aphid honeydew because it has a higher boiling point and hence is more difficult to remove in processing.

Once species verification has occurred, the judicious monitoring of populations should take place as soon as the pest has been identified as present in a field. The sampling protocol is as follows:

1. Designate a management unit of between 20-35 ha in size. Sample 15 leaves from 2 sampling sites within the management unit at least weekly.
2. Choose a plant to sample at least 10 m into the field avoiding plants disturbed by sweep netting or beat sheeting. Choose healthy plants at random along a diagonal or zigzag line moving over several rows taking 5-10 steps before selecting a new plant.
3. Choose a leaf from the 5th main stem node from the top of the plant as shown in Figure 3.



Counting the adults

- Keep shadow off the plant.
- Carefully turn the leaf over by the tip of the leaf blade or the petiole.
- Tally leaf as “infested” if it contains 3 or more whitefly adults and tally the leaf as uninfested if it contains less than 3.



Greenhouse whitefly adults hold their wings flat and appear 'heart' shaped when viewed from above.



Silverleaf whitefly adults appear narrow when viewed from above.

Figure 3.

Sample the fifth main stem node leaf.

Counting the nymphs

- Sample for nymphs within a 3.88 cm² (ten cent piece) disc.
- Place disc between the main veins on the left side.
- Count large (3rd and 4th instars) nymphs in area.
- Large nymphs are those that are visible to the naked eye and appear as flattened, egg-shaped discs or “scales.”

For more information on the management of silverleaf whitefly see the fact sheet ‘*Management of Silverleaf Whitefly*’ available from the Australian Cotton CRC website.



The armyworm caterpillar is sometimes found on young cotton but usually prefers other hosts.



Rough bollworm larva in a cotton boll.

Armyworms

This pest is often found in low numbers on young cotton, but usually prefers other weed hosts and cereal crops. Armyworms occasionally infest cotton seedlings heavily enough to cause defoliation. Armyworm should be sampled up until the start of squaring, recording numbers per metre.

Two species of armyworm are important:

Day feeding armyworm	<i>Spodoptera exempta</i>
Lesser armyworm	<i>Spodoptera exigua</i>

The lesser armyworm is often present in low numbers on young cotton.

Rough bollworms (*Earias huegeli*)

Rough bollworm is not common, but checkers should be aware of this pest from the time bolls exceed 1 cm in diameter, until the time when there are less than 5 bolls of this size per metre. Record the abundance of this pest per metre during normal sampling. If more than 2 per metre are found in a check during the susceptible stage of the crop then collect 100-200 large bolls as you walk from plant to plant during normal insect sampling. After the sample is collected, observe closely the exterior of the bolls. Cut open the bolls with frass or holes to check for the presence of rough bollworm larvae and count the number of bolls infested.

Thrips

Three species of thrips are common in cotton, *Thrips tabaci*, *Frankliniella schultzei* and *F. occidentalis*, all of which are pests as well as predators of mite eggs. Of these three species *F. occidentalis* has developed resistance to insecticides.

You only need to sample for thrips on pre-squaring cotton. Once the plants have more than 6 true leaves, sampling for thrips can be discontinued. Score the number of thrips (adults and nymphs) per plant. Check also for seedling damage, which should be taken into account in any decision to spray for thrips. When counting thrips check for the presence of nymphs. This indicates if the population is actively breeding in the cotton. This is important when checking plants that have had an insecticide seed treatment or an in-furrow insecticide treatment (granular or spray) at planting. On these plants there may be adults, due to constant immigration of thrips into the field, but no nymphs and little or no damage.

Points on thrips sampling

1. It is best to count from 20-30 plants separately. Score the number of thrips (adult and nymphs) on each plant. Use a hand lens to aid counting the smaller thrips larvae. Tease the terminal apart gently with a probe, such as a sharp pencil and the thrips larvae will move out and can be counted.
2. Thrips are often blamed for tipping out but are rarely the cause. For thrips to cause tip damage they must be in high numbers (> 30 per plant). It is generally possible to distinguish thrips damage from that of other pests such as *Helicoverpa*, tipworm or mirids. *Helicoverpa* or tipworm damage shows obvious signs of tissue removal and often a distinct ‘drill’ or bore hole can be seen. Tip damage by mirids will often

show no other signs of damage other than a blackening of the embryo leaves in the terminal without any leaf crumpling. Tip damage caused by thrips is typically associated with the presence of large numbers of thrips which will be very active if the terminal is disturbed. The small leaves around the terminal will also show extensive crumpling and blackened edges. Score tip damage taking note of whether the damage was from thrips or some other pest such as *Helicoverpa* or mirids.

The main effect of thrips feeding is distorted growth of leaves resulting in reduced leaf area. Plants can tolerate up to 80% reduction in leaf area until the 6 true leaf stage (leaves less than 1 cm in length) without affecting yield or maturity. Score estimated leaf area loss compared to that expected for an undamaged plant. As a rough guide if the average leaf size of a thrips damaged leaf is less than about 1 cm then leaf area reduction is often greater than 80%.

Thrips may buildup to quite high levels later in the season on cotton that has had very few insecticide sprays, especially if only selective insecticides are used. Adults will be seen in the flowers and the white or yellow nymphs can be found on the undersides of leaves in the upper canopy. These late season populations may cause damage to young leaves, resulting in distortion, but will also help to control mite populations. Control would only be justified if thrips were reducing growth dramatically, to the point where yield could be affected.



Young cotton damaged by thrips.

Jassids

There are at least two species found in cotton, the vegetable leafhopper (*Austroasca viridigrisea*) and the cotton leafhopper (*Amrasca terraereginae*). Jassids are usually only sampled on pre-squaring cotton, however in recent seasons the presence of very high numbers throughout the entire season has required season long monitoring. Jassids can be monitored using visual, beat sheet or d-vac techniques.

Score the number of jassids seen per metre. Jassids are very active so counts need to be made quickly.

Pink spotted bollworms (Pectinophora scutigera)

This pest occurs in coastal and Central Queensland only. The minute eggs are not easily found and therefore the presence of this pest is not detected until the larvae hatch and begin to tunnel in fruiting structures. To sample for this pest, split open 50 bolls to examine the inner boll wall for infestation and calculate the percentage infested.

Occasional Pests

There are certain pests of cotton which do not appear every season, but which may need control when they occur. Insects that fall into this category include wireworms, cutworms, bean root aphids, flea beetles, Rutherglen bugs, Redbanded shield bugs, plague locusts, and springtails. You should use your own discretion, or contact your local industry development officer for advice when deciding on management options.



The redbanded shield bug adult is an occasional pest found in cotton crops.

3.2.5 Early season plant damage

Cotton plants can recover from a degree of early season damage with no reduction in yield or delay in maturity. It is important to include an assessment of plant damage into pest management decisions because insect numbers alone may not give an accurate indication of the need for control. For instance, a vigorous healthy crop can tolerate more damage from pests, without yield or maturity being affected, than a crop with poor vigour (e.g. as a result of herbicide damage or water stress). On pre-squaring cotton it is important to assess leaf damage and tip damage.

Once squaring begins, fruit retention should be monitored in conjunction with pest sampling. This will indicate if pests such as mirids or *Helicoverpa* are causing a significant effect on fruit retention.

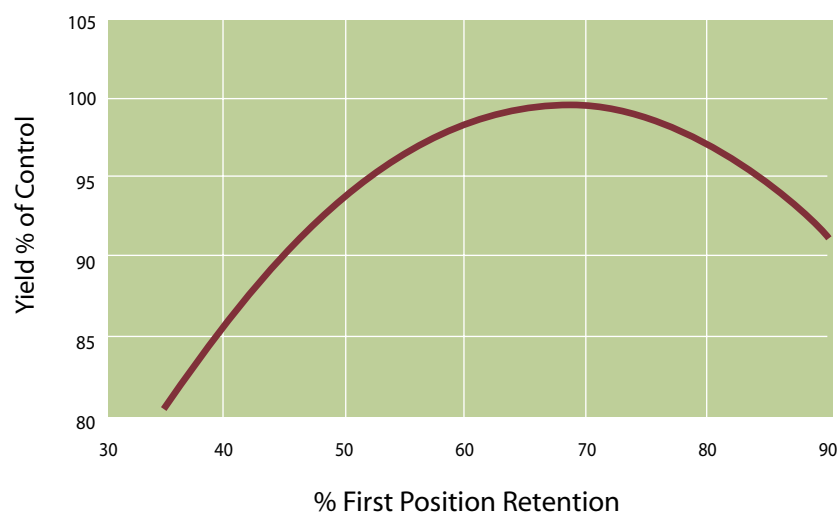
3.2.6 Monitoring tip damage

Monitor tip damage caused by different pests on pre-squaring cotton. Damage from thrips and *Helicoverpa* can be lumped together. Score the % of terminals with light damage (as caused by thrips, *Helicoverpa*, tipworm prior to entrenchment and light mirid damage) or heavy damage (entrenched tipworm or heavy mirid damage). Light tip damage is when only the growing tip has been damaged, whereas heavy damage is where one or two nodes below the terminal are also affected. Light mirid damage symptoms are typically blackened and withering terminals, while heavy mirid damage shows as the terminal and one or more nodes below the terminal also blackening, wilting and dying.

3.2.7 Monitoring fruit load

It is important to monitor crop growth rates and fruit development to avoid excessive periods of crop damage. Acceptable damage levels will vary depending on yield expectations and climatic conditions. Fruit load is a key aspect in determining crop yield and maturity. The loss of fruit during squaring and early flowering is less critical to yield than fruit loss later in the season. It is well documented that excessive early fruit loss can delay final maturity. However, it is also known that holding too much fruit can reduce crop growth, cause premature cut-out, and reduce yield. Crop yield and maturity is not affected if 1st position fruit retention is maintained around 60% at first flower.

Figure 4.
Retention at flowering vs control
yield. Summary from Commercial
scale trials.



Decisions about the need to control pests should therefore take into account both pest numbers and plant fruit load. If retention data indicates that fruit load is too low then it may be necessary to lower the pest threshold. Alternatively, if retention is too high then it may be necessary to raise the pest threshold. This will allow some pest damage and help balance vegetative growth and fruit development. This will also avoid yield loss due to premature cut-out. Such an approach treats the pest threshold as dynamic, that is, it varies according to how the plant's fruit load is developing.

Assessing fruit load in conjunction with regular insect monitoring provides significant benefits:

- An assessment of the effects of mirids on fruit load (especially in Bollgard II® crops) and the effects of combinations of mirids and *Helicoverpa* – particularly where *Helicoverpa* species are just below the threshold.
- Complements insect checking, especially for pests that are difficult to detect during regular insect checks.

There is a range of tools available to assess fruit load. These include:

- Total retention of first position fruit

- Top 5 fruiting branches (use this method up to 10 days post flowering)
- Fruiting factors (count first and second position fruit on major fruiting branches)

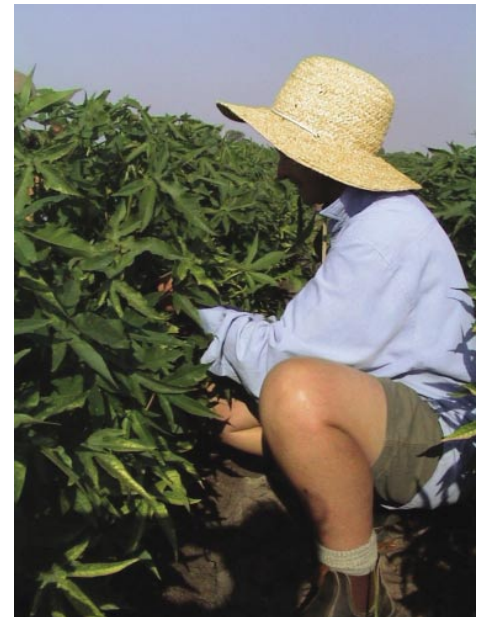
3.2.8 Monitoring first position fruit retention

Monitoring first position fruit retention is a technique that is best used early in the season during squaring and prior to or just after flowering. It is quicker than total fruit counts and can provide an early sign of insect damage. Fruiting factors can be used throughout the season and allow total fruit load to be monitored. Fruiting factors should be used when first position retention falls below recommended levels (i.e. 50% to 60%), to ensure excessive fruit loss has not occurred, or in situations where a crop is tipped out and retention is difficult to determine.

Fruit retention (first position or top 5)

Six step monitoring technique (also see Figure 5.)

- 1) Only monitor first position fruit retention
 - use either top 5 or all fruiting branches
 - first fruiting branch is identified by the branch where the first position leaf is unfolded
- 2) Monitor plants within a linear metre in 3 to 4 locations per field
 - do the same plants used for insect checks: do not select individual plants at random
- 3) Monitor both tipped and non-tipped plants
- 4) Monitor only the dominant stem, not vegetative branches
- 5) Monitor at least 30 plants per field (3 - 4 m)
 - a four fold error can occur when only 20 plants are selected
 - increase sample size if crop is very uneven
- 6) Monitor every 7 days and / or before spray decisions
 - it is important to monitor the level of fruit loss regularly before insect damage becomes excessive, especially if pest combinations are present, e.g. mirids + *Helicoverpa*
 - start monitoring after the first week of squaring



Assessing crop fruit load is an important tool to check crop progress and evaluate if there has been excessive fruit loss.

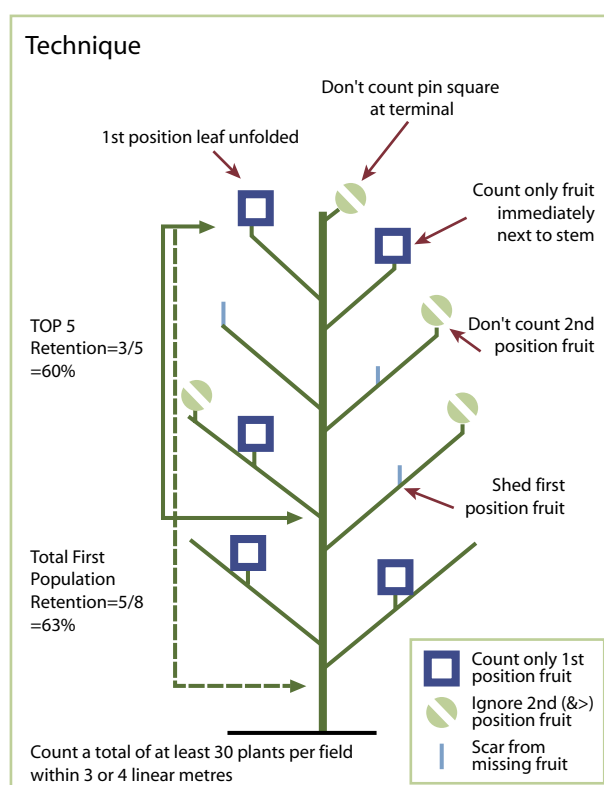


Figure 5.

A technique for checking fruit retention.



Boll numbers will vary according to variety, stage of growth and yield potential.

Interpreting retention data

Growers should aim to have first position fruit retention of 50-60% by first flower. Lower retention (< 50%) increases the risk that yield or crop maturity will be affected. However, very high fruit retention, in excess of 80%, may also be associated with a yield penalty as the plants use their resources to fill the bolls they have set rather than continuing to grow and set further fruit. For the first 5 fruiting branches on the plant, first position fruit retention can be as low as 30% without affecting yield or maturity. Such levels should trigger close monitoring as the plant grows, as retention must increase in order to achieve 50-60% at first flower. If fruit retention is too low (< 50% retention over two consecutive checks) it is important to take action to ensure that it recovers to acceptable levels by flowering. Firstly, identify and where possible rectify possible causes of low fruit retention. These could include factors such as weather, waterlogging, water stress or pests. Secondly, the combined damage of several pests, each below threshold, may also cause low retention. Therefore reduce pest thresholds to half the standard level and control those pests exceeding the reduced threshold using the most selective option available. As retention recovers, return to the standard pest thresholds.

Final retention at maturity

Boll numbers will vary according to variety, stage of growth and yield potential. At the end of the season a crop will hold less than 50% of all possible fruiting sites. First position retention will vary from 50% to 70%. Variety and boll size will also affect final yield.

3.2.9 Monitoring fruiting factor

This method is used to check total fruit numbers. When retention falls below recommended levels a total fruit count should be conducted to ensure excessive total fruit loss has not occurred. Also from 10 to 14 days after flowering, monitoring the first position fruit retention may be less relevant than fruit counts. To allow a rapid interpretation of the fruit counts, a fruiting factor has been developed which considers both fruit counts and the number of fruiting branches.

To determine the fruiting factor for a crop, divide the fruit count by the number of fruiting branches.

$$\text{Fruiting Factor} = \frac{\text{Total fruit count per metre}}{\text{Total number of fruiting branches per metre}}$$

To save time, only count first and second position fruit (squares and bolls) from the main stem, and the first dominant vegetative branch. In irrigated crops this should account for 90% of the fruit to be picked.



Flowering is a key period for measuring fruiting factors.

Fruiting factors change throughout the growing season as plants set more fruit. During squaring, values of 0.8 to 1.0 are normal for high yielding crops. The fruiting factor will increase throughout flowering as the crop produces a large number of squares. After peak flowering and as the crop matures, the fruiting factor will decline. This coincides with the natural reduction in fruit numbers. At maturity the fruiting factors will approach a value of 1.0, which represents the natural maximum fruiting load that plants can carry through to yield.

Flowering is a key period for measuring fruiting factors. Values between 1.1 and 1.3 will provide optimum yield potential. Values less than 0.8 or greater than 1.5 can cause yield reductions.

Table 5. General guide to using fruiting factors throughout the season

Stage of growth	Fruiting factor
Pre flowering	0.8 - 1.0
Flowering	1.1 - 1.3
Peak Flowering	1.3 - 1.4
Boll maturity	1.0

Table 6. General guide to using fruiting factors at first flower

Fruiting factor at flowering	Impact on yield and maturity
Less than 0.8	High risk of yield decline and maturity delay (particularly in cooler regions)
1.1 to 1.3	Optimum for yield
More than 1.5	Risk of premature cut-out and yield decline

3.2.10 Bollgard II® sampling and management

Effective sampling of Bollgard II® is critical for good management and should be similar in intensity and frequency to conventional cotton varieties.

Bollgard II® cotton provides very effective control of *Helicoverpa* spp. and a range of other lepidopteran pests. However, the Cry proteins in Bollgard II® (Cry 1Ac and Cry 2Ab) do not control other pests. Furthermore, the reduction in spraying against *Helicoverpa* spp. allows some pests, formerly controlled by these sprays, to become more of an issue.

Amongst these pests, the green mirid is potentially the most difficult to manage, because numbers can change in a short period and damage can occur quickly. The green mirid will cause tip damage to pre-squaring cotton, and square and boll damage in mid season cotton. Sprays to control mirids are generally broad spectrum, or very expensive selective options. Spraying mirids therefore may cause a reduction in beneficial insects, which may in turn allow secondary pests such as mites, aphids or silverleaf whitefly to surge. In addition, some sprays applied against mirids such as organophosphates may also select for resistance in other pests, such as aphids.

Sampling Bollgard II® cotton to monitor mirid populations and the abundance of other pests as well as plant damage is therefore critical for maximising the potential of this technology.

Bollgard II® cotton must be monitored regularly throughout the season for *Helicoverpa* and other pests. *Helicoverpa* spp. damage can occur in pre-flowering crops under conditions of reduced Bollgard II® plant efficacy or heavy insect pressure. It is important to check the Bollgard II® cotton at least every 3 days to identify such damage, and to monitor fruit retention in the same way as for non-transgenic cotton.

The thresholds for *Helicoverpa* on Bollgard II® are 2 very small or small larvae (> 3 mm) / m or 2 total larvae (> 3 mm) per m where the threshold must be exceeded on two consecutive checks, or 1 medium or larger larva per m where the threshold applies on the first check. Management of damage and other pests should follow the same guidelines as conventional cotton. The predator / beneficial to pest ratio approach can also be applied effectively. The ratio will remain well above 0.5 while the Bollgard II® is controlling *Helicoverpa* effectively. As control declines the use of the predator / beneficial to pest ratio becomes more critical as a means of including the value of beneficials in decision making. For more information refer to the latest 'Cotton Pest Management Guide'.

3.2.11 Pest abundance and damage thresholds

Pest and damage thresholds are a core component of integrated pest management (IPM). They provide a rational basis on which to make pest control decisions. Thresholds can be based on the abundance of the pest, the damage they cause, or a combination of both. The economic importance of an individual pest species may vary with the development of the cotton plant. It is convenient to divide crop development into three phases, and where appropriate, apply specific thresholds to each phase. Effective use of thresholds is dependent upon accurate, objective sampling to provide reliable estimates of pest and/or damage levels.

The pest and damage thresholds are shown in Table 8, and the following information is specific for each pest.



Yellow night stalker feeding on a green mirid adult.



Pest checks should be made over the whole plant including in terminals, squares, flowers and bolls.



A cotton boll damaged by a *Helicoverpa* larva.

Helicoverpa

CottonLOGIC supports the *Helicoverpa* development model which can be used to estimate the development of a given egg and larval population over the next 3 days, taking into account estimated natural mortality levels for the time of the season.

Egg thresholds

The egg threshold for conventional cotton is 5 brown eggs / m from flowering through to harvest, with no threshold based on white eggs. For Bollgard II® cotton there is no threshold based on white or brown eggs as the Bt proteins must be digested by the larva in order to take effect.

When deciding if an ovicide spray is required, the predator / beneficial to pest ratio should also be taken into account to allow for natural control from predators and parasites. Environmental conditions should also be considered, especially early season when plants offer little protection against hot dry winds or rain which can result in high egg mortality.

Larval thresholds

Beneficials can significantly reduce larval numbers, often to below threshold levels. Control decisions should take into account the predator / beneficial to pest ratio as an indication of the likely effectiveness of beneficials in reducing larval numbers. Plant damage levels should also be considered. The survival of *Helicoverpa* larvae on young cotton is often quite low. However if growing conditions are warm, plants will tolerate being tipped out twice by the surviving *Helicoverpa* without affecting yield or maturity. If conditions are cooler and growth slower, the *Helicoverpa* tip damage threshold should be reduced to once per plant.

The larval threshold until cut-out is a total of 2 / m. For conventional cotton this is based on a single check, but for Bollgard II® cotton it is based on 2 consecutive checks. The threshold for conventional cotton after 15% of bolls are open can be raised to 5 total larvae per metre or 2 medium / large larvae per metre. This threshold is preliminary and should be applied cautiously. For more information see Table 8, '*Pest threshold summary*'.

The first sign of two-spotted spider mite damage is bronzing of the upper leaf surface near the petiole.



Mites

A nominal threshold of 30% of leaves infested is used from seedling emergence up to 20% of bolls open, after which mites will no longer affect yield. Alternatively mite control can be based on the predicted yield loss that a given mite population is likely to cause. Information for predicting yield loss due to mites is available in the *Cotton Insect Management Guide* and in CottonLOGIC. The '*Managing mites in cotton*' CRC research review is available on the Australian Cotton CRC website.

Mirids

Mirids are a pest of the vegetative seedling stage and of the early squaring stage (up to 2-3 weeks after squaring begins). The thresholds are based on accurate sampling for mirids using either visual counts or a beat sheet. The threshold for visual sampling for cooler regions is 0.5 mirids per metre (adults and nymphs combined) while in warmer regions it is 1 per metre. These thresholds should be multiplied by 3 for beat sheet samples. It is critical that these thresholds are used in combination with tip damage monitoring and fruit retention sampling (see '*Early season plant damage*' in this objective). If retention is above 60% it is possible to tolerate higher numbers of mirids without suffering a reduction in yield or delay in maturity.

Tip damage from mirids can vary widely in severity. When mirids feed they inject saliva into the plants which contains chemicals known as pectinases. These chemicals can delay the recovery of plants and sometimes cause multi-branched 'crazy' cotton. Light damage from mirids may result in light tipping out that has no effect on yield and little (if any) effect on maturity. For this type of damage a threshold of 50% of plants tipped out can be used. However, progressively heavier mirid damage will result in more of



Multi-branched cotton caused by mirid feeding.

the plant being affected, with nodes below the terminal being increasingly damaged. Heavy damage may result in the death of the growing terminal and the 2-3 nodes below it. On seedling cotton, this can result in death of the terminal back to the cotyledons, resulting in plants that never recover. A threshold of 20% of plants heavily tipped out by mirids should be used.

Aphids

The cotton aphid threshold is 90% of plants infested until 1% of the bolls are open, after which it drops to 50% infestation. There is no honeydew threshold until 1% of bolls are open. After 1% of bolls are open and honeydew is present, the aphid threshold is reduced to 10% infestation. Check field borders and spray them separately when necessary.

The threshold for the green peach aphid is 25% of plants infested in phase 1 as it may be a problem early season, however populations normally decline in hot weather. Some cotton aphid and green peach aphid strains are resistant to organophosphates and carbamates. This needs to be taken into account when control options are being considered. See the insecticide resistance management strategy included in the '*Cotton Pest Management Guide*'. For more information on aphid management visit the Australian Cotton CRC website.



As seasonal temperatures increase, green peach aphid populations usually decline.

Thrips

The need to control thrips is based on both the thresholds for thrips numbers and damage being exceeded. Control is justified if there are 10 or more thrips per plant and the reduction in leaf area due to thrips is greater than 80%. Control is justified if there is a reduction in leaf area of more than 50% once the plant has reached the 6 true leaf stage. Thereafter thrips are unlikely to affect the yield or maturity of cotton crops. These levels of leaf damage will need to be exceeded before thrips cause tip damage. If conditions are cool or the plant has another setback, such as herbicide damage, then the thresholds can be reduced.



It is important to assess damage from thrips as it is often cosmetic and plants usually recover without loss of yield or delay in maturity.

Rough bollworms

Susceptibility to rough bollworm starts when there are more than 5 bolls / m over 2 weeks old (> 1 cm in diameter). Susceptibility ceases when there are fewer than 5 growing bolls / m less than 2 weeks old.

Green vegetable bugs (GVB)

Cotton is attractive to GVB from boll-set onwards, and crops should be inspected using the beat sheet technique. The GVB can cause significantly more damage to bolls less than 21 days old, particularly those less than 10 days old. GVB instars 4, 5 and adults inflict the same amount of damage. Instar 3 does half the damage of an adult, and a cluster (more than 10) of first and second instars does as much damage as one adult. Table 7 provides a description of each instar for correct identification and determination of the correct threshold. For more information and pictures of these GVB instars go to '*The Pest and Beneficial Guide*' on the Australian Cotton CRC website. When managing GVB populations it is also important to monitor fruit retention. Retaining 50-60% first position fruit by first flower is an ideal target.



Green Vegetable Bug 5th instar nymphs cause the same amount of damage as adults.

Table 7. Description of Green Vegetable Bug nymphal stages

Stage / Instar	Instar Length (mm)	Description
1	1	Predominantly orange
2	2	Black with 1 or 2 white spots
3	4	Mosaic pattern of green black and red spots
4	7	More green spots, wings begin to develop during late 4th instar
5	10	Spots start to diminish to green, wings well developed
Adult	15	All green with wings

As GVB does not damage pre-squaring cotton, thresholds only apply to phases from squaring onwards. For visual counts the threshold for GVB adults is 0.5 / m and for beat sheet counts it is 1 / m. The damage threshold for phases 2 and 3 is 20% damage to small bolls (bolls around 14 days old).

Armyworms

For visual counts, the thresholds for small larvae (< 7 mm) are 2 / m or for large larva (> 7 mm) 1 / m. These thresholds only apply for the first crop development phase.

Pink spotted bollworms

The threshold for pink spotted bollworm is based on the infestation as determined by examining inner boll walls.

Tipworm

Damage due to exposed small tipworm larvae is similar to that of *Helicoverpa*, and plants will tolerate this damage as described above. Once tipworm become entrenched they bore down into the main stem and the effects of their feeding may damage several nodes below the growing tip. The tolerance of cotton to this type of damage is not well understood. Anecdotal evidence suggests that all plants can be damaged by entrenched tipworm once without affecting yield or maturity. This depends on crop growth, with poorly growing crops being more affected, in which case half this threshold should be used. Repeated events of damage from entrenched larvae may not affect yield but may cause delays of 1 – 3 weeks depending upon severity. This exposes late maturing cotton to potential infestations of resistant *H. armigera* and increasing insecticide costs. However, tipworm is rarely abundant enough to be an economic problem. This is because seasonal conditions through the winter and spring are rarely ideal for a sufficient buildup of the primary hosts of tipworm (marshmallow or anoda weed). This restricts population buildup and the probability of this pest reaching damaging levels in cotton. It should be noted that Bollgard II® cotton provides good control of tipworm.

Silverleaf whiteflies (SLW)

The most effective control option for SLW is the use of insect growth regulators (IGRs). These insecticides prevent populations from building up, but have little effect on predators and parasites that also help control this pest. Effective use of the IGRs requires application at threshold. Earlier applications may not provide effective control and later applications may be too late with the IGR not controlling enough SLW to prevent later buildup.

Threshold for use of insect growth regulators

0.5 – 1 Nymph / Leaf Disc

and

3 – 5 (39 – 57%) Adults / Leaf

Use the decision matrix:

Decision Matrix for Insect Growth Regulator use in Cotton, modified from *Management in Arizona Cotton* 1996.

		Whitefly Adult Levels	
		Fewer than 3/leaf	At least 3-5/leaf
Whitefly Adult Levels	Presence/absence large nymphs of 30 discs sampled	Wait and re-sample in 3-7 days	Re-sample in 3 days OR apply IGR
	Actual numbers/leaf disc (average)	Re-sample in 3 days OR apply IGR	Apply IGR

Threshold for use of other chemistry

5 (57%) Adults / Leaf

No use of other chemistry based on nymphal counts

For more information please refer to '*Management of Silverleaf Whitefly in Australian Cotton*', which can be found on the Australian Cotton CRC website and is also available from the TRC.

3.2.11.1 **Table 8.** Pest threshold summary (visual counts)

Insect Pest		Planting to flowering (1 flower per m)	Flowering to Cut-out (1 open boll per m)	Cut-out to Harvest	
				Up to 15% open	After 15% open
Helicoverpa/m					
W. eggs		-	-	-	-
B. eggs		-	5	5	5
Total Larvae		2	2	3	5
M.+L. Larvae		0.5	1	1	2
Tip damage (% of plants affected)		100-200%	-	-	
				NB Helicoverpa control can cease at 30-40% bolls open	
Tipworm					
Larvae/m		1-2	-	-	
Tip damage% of plants affected	Not entrenched	100-200%	-	-	
	Entrenched	50-100%	-	-	
Mirids					
Adults & nymphs/m	Cool region	0.5	0.5	0.5	
	Warm region	1	1	1	
Tip damage% of plants affected	Heavy	20%	-	-	
	Light	50%	-	-	
Mites (% of plants infested)		30% of leaves infested. However, thresholds based on potential yield loss are available. Yield loss is estimated using time of infestation and rate of increase, see 'Cotton Insect Management Guide'. See sampling guidelines for further details of mite management on seedling cotton. Mites will not effect yield after 20% of bolls are open.			
Cotton aphid (check species)					
% of plants infested		90%	90%	50% (10% if honeydew present)	
Honeydew present		-	trace	trace	
Green peach aphid		25%	May be a problem early season, populations normally decline in hot weather.		
Armyworm					
Large larvae/m		1	-	-	
Small larvae/m		2	-	-	
Rough bollworm					
Larvae/m		2	3	3	
Damaged bolls (%)		-	3%	3%	
Loopers/m		-	20	50	
Thrips					
Adults & larvae per plant		10	-	-	
Damaged (reduction in leaf area)		80%	-	-	
Green vegetable bug adult/m					
Visual		-	0.5	0.5	
Beat sheet, or		-	1	1	
Damage to small bolls (14 day old)		-	20%	20%	
Jassids (leaf hoppers)/m		50	-	-	
Pink spotted bollworm					
% bolls infested		-	5	5	
Fruit retention monitoring		50-60% first position fruit retained by 1st flower (1 flower/m)			
Fruiting factor monitoring		see tables 5 and 6			



Tipworm larvae tunnel into the terminal causing multiple branching.



Leaves damaged from thrips take on a silvery, bleached appearance and younger leaves become distorted in shape.

Table 8. continued*Helicoverpa* Thresholds for Bollgard II® Cotton (visual counts)

<i>Helicoverpa</i> /m	All season
W.Eggs	-
B.Eggs	-
Total Larvae (Excluding larvae<3mm)	2/metre over 2 consecutive check
M&L Larvae	1/metre on the first check

NB Thresholds for other pests and damage are as above

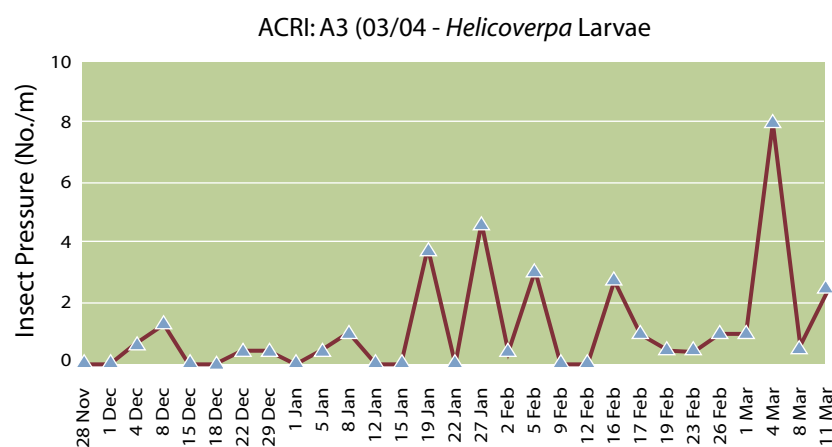
3.2.12 Recording and reviewing data to make decisions

Regular and frequent checking provides an overview of what is happening in a field in relation to pest and beneficial abundance and development. A generalised decision process using pest and damage data is shown on page 33. It is generally not possible to make a decision about whether control is needed based on just one check. In most instances, a decision is made on the basis of trends in population growth and the impact of beneficials (loss of eggs, larvae and nymphs). For example, over several consecutive checks above threshold numbers have been found, but *Helicoverpa* eggs and larvae have been reduced to below threshold numbers from one check to the next. Delaying a control decision on an above threshold population in a current check would be justified as there is evidence of sufficient mortality due to beneficials or weather conditions. Unless you look at the data over a series of checks you will not get this information.

While data is recorded on individual sheets or cards for each checking date, it is useful to find a way to review this information on a weekly or fortnightly or even season long basis to get a feel for trends. This type of reviewing can be done using a spreadsheet, or using CottonLOGIC, by graphing the check data for several checks as shown in Figure 6.

Figure 6.

Seasonal graph produced by CottonLOGIC showing the seasonal trend in abundance of *Helicoverpa* spp.



The strength of reviewing the data in this way is that it provides an idea of whether *Helicoverpa* eggs and larvae, tipworms, rough bollworms, or populations of mirid adults and nymphs are surviving and developing, and whether their numbers are higher or lower than you would expect. This information builds confidence in deciding whether control can be delayed. It also provides the best measure of whether beneficials are actually having an impact on the pests. This is particularly the case for beneficials that are often not accounted for in the predator to pest ratio, like egg and larval parasitoids which prevent eggs and larvae developing, but are not easily counted in the field (see section 'Use of parasitoids in spray decisions - using the beneficial insect to pest ratio' in objective 3).

Reviewing data over consecutive checks is essential for making decisions about the management of *Helicoverpa* in Bollgard II® crops, as the Bt toxin needs to be ingested before the larva is controlled. Hence if the larvae population is over the threshold on a given check, then chances are that a large proportion of these will ingest the toxin and die before the next check.

Thresholds are provided in Table 8, the ‘*Pest threshold summary table*’.

Example decision making process for pest management

SAMPLING		
<i>Monitor plant damage:</i> • Tip Damage • Fruit Retention	<i>Monitor pest densities:</i> • Caterpillars • Mirids and GVB • Mites • Aphids • Whitefly	<i>Monitor beneficials:</i> • Predators • Parasites
ASSESSMENT		
• Check damage thresholds • Consider assessing fruiting factor • Apply Early Season Diagnostic (ESD) tool	• Check pest thresholds • Track pest trends	• Check beneficial to pest ratio • Check parasitism rates
DECISION		
<i>Does crop need protection?</i> <i>Consider:</i> - Is it ahead of schedule and can tolerate some damage? - Is poor retention due to pests or other factors?	<i>Are pests over, or close to the threshold?</i> <i>Consider:</i> Are pests increasing, static or decreasing? For pests such as mites, aphids and whitefly static or decreasing populations often indicate good predation levels.	<i>Are predator numbers or parasitism rates too low to control pests?</i> <i>Consider:</i> - Proximity to sources of beneficials. - Other pests present that beneficials may be controlling.
NO... Come back later and check again in two days		
YES... Choose an option		
ACTION		
<i>Consider application issues:</i> - Wind, temperature, sensitive areas, Bt refuge regulations - Impact of a spray on neighbours and nearby crops	<i>Check IRMS regulations</i> <i>Consider:</i> - Available options - Consecutive sprays (resistance in target and other pests) - Application limits - Other pests present that may require control or be ‘flared’ i.e. mites, aphids, whitefly	<i>Check impact table</i> <i>Consider:</i> - Impact on beneficials - Potential to flare pests - Using food sprays or biological insecticides or manipulating lucerne trap crops to restore the predator to prey ratio
EVALUATION		
<i>Continue to monitor plant damage.</i> • Is the plant on track? • If not, is it pest related or due to other factors.	<i>Resample</i> • Is the pest below threshold? • If not, why not – poor application, resistance, insufficient time (eg miticides, whitefly insect growth regulator) • Formulate new control options in relation to, efficacy, resistance and impact on beneficials.	• Monitor beneficials and calculate predator to prey or beneficial insect to prey ratios. • What impact did the control have on beneficials – note for next time.

3.2.13 Computerised decision support for pest management

From the CottonLOGIC suite of computerised decision support tools, EntomoLOGIC supports a range of sampling methods including those described earlier in the section ‘sampling techniques’. It also provides an effective means of recording and storing pest and crop data and with a number of facilities for charting pest abundance. EntomoLOGIC also supports beneficial sampling and the calculation of predator to pest ratios.

EntomoLOGIC provides the tools to help:

- Verify your crop management decisions
- Achieve your best management practice goals
- Manage and analyse your farm records
- Manage your time and resources effectively
- Assist with pest management decisions by predicting pest populations

- Analyse your crop management decisions using the graph generator
- Optimise your nitrogen fertiliser use
- Keep track of your crop development with plant mapping
- Identify cotton pests and beneficials with pictures and information
- Streamline the data entry of farm records by electronically ordering sprays.



A waterproof pouch can be used to protect the handheld device.

CottonLOGIC for Palm OS® handhelds (including EntomoLOGIC):

- Takes decision making into the field
- Improves insect data management and integrity
- Streamlines data collection, storage and analysis
- Runs models of pest development
- Generates in-field reports for on the spot printing
- Gives you the opportunity to use your time more productively

To obtain the CottonLOGIC software, or for more information contact the coordinator, TRC (02) 6799 1534.

3.2.14 Using the Early Season Diagnostic (ESD) tool

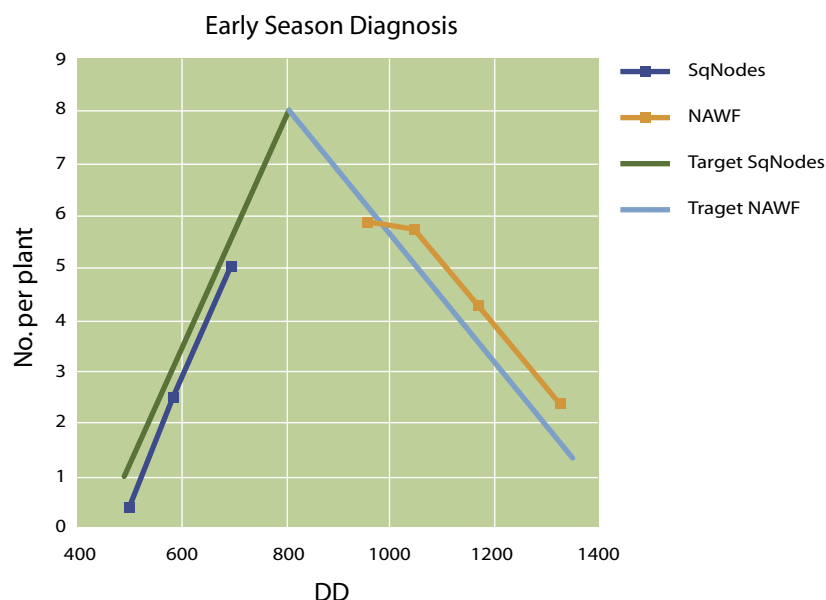
The Early Season Diagnostic (ESD) tool has been developed to help growers identify growth problems in their cotton crop. The ESD tool can help growers achieve optimal crop growth, maturity and yield. The system is based on graphically comparing the observed crop development data with a potential or target line. This target has been generated from data collected over many years of research where growth has been under non limiting conditions.

During the period after squaring (500 DD) and before flowering, weekly measurements of the average number of squaring nodes per plant and the number of DD after sowing are entered into the ESD tool. After flowering and through to cut-out (3-4 NAWF), weekly measurements of the average number of Nodes Above the White Flower (NAWF) and the number of DD after sowing are entered into the tool.

The Day Degrees (DD) and Node Counts are graphed and displayed against a crop development rate as shown in Figure 7 to help determine how well the crop is growing. Measurements well above the potential line are rare but may need to be managed to avoid early cut-out. Measurements well below the potential line indicate a problem with the crop development that may require a management solution (e.g. water or fertiliser).

Figure 7.

An example of the ESD output from a crop that is growing well (Data: Jenelle Hare, DPI&F Queensland)





3.3 Objective 3 - Beneficial insects - use them don't abuse them!

3.3.1 Introduction

Predatory and parasitic (beneficial) insects consume pests and other insects in order to survive, develop eggs and / or produce offspring. The most common predators found in cotton farms feed on a wide range of pests and are therefore classified as general predators. In contrast, parasitoids are specific to the insect or stage of insect they attack. For example, *Trichogramma* is an egg parasitoid of *Helicoverpa* spp., whereas *Microplitis* is a parasitoid that only attacks *Helicoverpa* spp. larvae. Predators and parasitoids can considerably reduce pest numbers thereby reducing the need to control them using insecticides. The abundance of beneficial insects is affected by food resources, mating partners, overwintering sites, shelter, climatic conditions and insecticide sprays. For an IPM system to work, it is important to know which species are present in a crop, how abundant they are and what pests they attack. Such information is important in evaluating their potential effect on a given pest situation, and in the choice of control measures if required, to conserve the populations.

There are a range of techniques to conserve and enhance beneficial insect activity on cotton farms, including:

- Use of the predator / beneficial to pest ratio in spray decisions
- Use of parasitoid activity in spray decisions (i.e. incorporating parasitism into the predator to pest ratio to become the predator / beneficial to pest ratio)
- Use of beneficial insect attractants (food sprays)
- Use of lucerne strips or blocks to conserve beneficials and manage mirids
- Use of refuge or nursery crops to conserve parasitoids
- The release of *Trichogramma* in cotton crops
- Tolerating non-economic early season damage
- Use of insect resistant transgenic cotton varieties
- Appropriate use of insecticides

For more information on the conservation of predatory and parasitic insects, refer to the 'Integrated Pest Management Booklet' in the 'Australian Cotton Best Management Practices' manual, and for identification of predators and parasites see the 'Pest and Beneficial Guide' on the Australian Cotton CRC website.

3.3.2 Use of the predator to pest ratio in spray decisions

Using the predator to pest ratio allows the grower to incorporate the activity of predatory insects into spray decisions. The calculation of the ratio includes *Helicoverpa* eggs and very small (VS) plus small (S) larvae per



The Red and Blue beetle is a common predator of *Helicoverpa* spp. eggs, small larvae and other slow moving insects such as aphids.



Helicoverpa brown eggs are collected to determine levels of egg parasitism.

visual metre assessed using standard visual sampling, but does not include medium (M) or large (L) larvae since many of the common predatory insects are not effective on these stages. Total predators per metre assessed using a standard visual check or the equivalent with other techniques, should also be used in the calculation. However, to be confident in the ratio, at least 3 insects of the most common predators within the families Coccinellidae, Melyridae, Nabidae, Lygaeidae, Reduviidae, Chrysopidae, Hemerobiidae and Pentatomidae should be present in a 20 metre d-vac sample or 3-5 metre visual checks. For more information on these families refer to the section ‘*Sampling beneficial insects and spiders*’ in objective 2.

The predator to pest ratio is calculated as:

$$\frac{\text{The number of predators per metre}}{(\text{Helicoverpa eggs} + \text{larvae (VS + S)})}$$

If this ratio is 0.5 or higher then predators will generally provide effective control of *Helicoverpa* spp. If the ratio is less than 0.5 then there are a range of options that can be taken, see ‘*Use of beneficial insect attractants (food sprays) to conserve and enhance beneficial insects*’ in this objective.

3.3.3 Use of parasitoids in pest management decisions - using the beneficial to pest ratio



A female *Trichogramma* wasp lays an egg inside a *Helicoverpa* egg.

Some parasitoids are particularly effective against pests and every effort should be made to consider parasitism levels when making pest management decisions. This is best achieved by assessing the levels of pest parasitism. For example, if you suspect that *Trichogramma* are active in your region then you should collect brown *Helicoverpa* eggs and determine the levels of egg parasitism. If you find high levels of egg parasitism then you may choose not to spray, or you may decide to use a ‘softer’ insecticide to manage the hatching of *Helicoverpa* larvae. In this way you have incorporated an assessment of parasitism into your spray decision. For further information on determining levels of egg parasitism, refer to the section ‘*Sampling and determination of Trichogramma parasitism in cotton farms*’.

The beneficial insect to pest ratio

The predator to pest ratio does not incorporate parasitoid activity, particularly egg parasitism in the calculation. Subsequently, for a grower to use both predators and parasitoids (especially *Trichogramma*) in spray decisions, the level of egg parasitism (i.e. percentage egg parasitism) should be deducted from the *Helicoverpa* spp. eggs per visual check before the predator to pest ratio is calculated. In doing so, the predator to pest ratio then becomes the “*beneficial to pest ratio*”.

Thus the beneficial to pest ratio is calculated as:

$$\frac{\text{Total number of predators per visual metre}}{(\text{Helicoverpa eggs per metre} - (\% \text{ egg parasitism}) + \text{larvae (VS + S)})}$$

If this ratio is 0.5 or higher then predators and parasitoids will generally provide effective control of *Helicoverpa* spp. Percentage parasitism used in the calculation should be based on the trend for parasitism on the farm. Parasitism levels in the previous or (last 2-3 days) count should be used to calculate the beneficial to pest ratio in the next check.

Example: In a grower or consultant’s check the following were recorded:

- Total predators per visual metre or beat sheet equivalent = 20
- No. of *Helicoverpa* eggs per metre = 40
- No. of VS+S per metre = 1.2
- Percent egg parasitism from previous check = 50%
- Beneficial to pest ratio (including trend of parasitism):

$$= 20 \text{ predators} \div (40 \text{ eggs per metre} - (50\% \text{ of } 40 \text{ eggs} = 20 \text{ eggs}) + 1.2)$$

$$= 20 \div (40 - 20) + 1.2$$

$$= 20 \div 21.2 = 0.94 \text{ which is higher than } 0.5.$$

This means, the system is working well and the grower need not apply any control for *Helicoverpa* spp. at this stage.

In contrast, if a predator to pest threshold was used for decision making, the ratio based on the above check would have been:

$$\text{Predator to pest ratio} = 20 \div 41.2 = 0.49.$$

This means that it is unnecessary to spray, but since the ratio is lower than 0.5 and *Helicoverpa* population is predominantly eggs, options to enhance predator numbers would be recommended. This could include the application of a yeast-based food spray to attract more predators, or slashing a beneficial nursery crop to encourage predators to move to the cotton crop.

Including the impact of egg parasites therefore allows for more accurate pest management decisions.

Calculating the beneficial to pest ratio should not include medium and large larvae because predators and parasitoids are not effective on these stages. The number of adult *Trichogramma* parasitoids per metre is not included directly in the calculation because *Trichogramma* adults are very tiny and cannot be counted visually.

3.3.4 Sampling and determination of *Trichogramma* parasitism

It is very important not to spray *Helicoverpa* based on egg counts alone. Parasitism of *Helicoverpa* spp. eggs by *Trichogramma* on cotton and other crops can result in significant egg mortality and reduce the number of larvae hatching from these eggs even at high *Helicoverpa* spp. egg densities. Consequently, growers should make every effort to consider parasitism levels when making spray decisions.

Egg collections

The best way to determine the level of egg parasitism in your crop is to collect eggs and wait to see how many hatch and how many turn jet black. It is important to only collect brown eggs (eggs that are about two days old) when assessing parasitism. White eggs may have just been laid and *Trichogramma* may not have had sufficient time to find them. If you collect white eggs you will generally underestimate the levels of egg parasitism.

In cotton you should randomly collect at least 20 eggs on leaves or squares from different plants in a field. In vegetative sorghum and maize you can also collect eggs on leaves. For heading sorghum it is best to sample pre-flowering heads just after they have fully emerged from the boot, i.e. before the first yellow flowers appear on the top of the head. To collect the eggs from sorghum you remove the head using secateurs and spin it into a plastic bucket. The eggs will fall into the bucket and can be stored in a container to be sorted later. Try to collect at least 50 eggs from a minimum of ten heads.

It is also important to keep the eggs cool in an esky after you have collected them because exposure to high temperatures, e.g. in a car glove box or on a dash board may kill them.

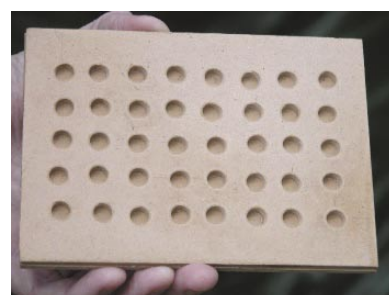
Storing Eggs

Helicoverpa larvae are cannibalistic, and caterpillars that hatch from unparasitised eggs will eat nearby eggs if they are free to roam. To prevent this, the eggs must be isolated individually in a multi-celled egg tray. To make your own egg tray use 3 mm thick craftwood and drill a series of 6 mm diameter holes in one piece and glue this to a solid piece of craftwood.

To transfer the brown eggs to the egg tray use a fine paint brush. Be careful when removing the eggs from the leaves as they are fragile and can be easily damaged. When the egg tray is full, cover the holes securely with sticky tape to prevent the larvae that hatch from unparasitised eggs moving into nearby cells and eating eggs. Store the egg trays at room temperature, e.g. in your office or kitchen, but don't leave them in the shed as it may get too hot during summer.



It is important to consider natural levels of *Helicoverpa* egg parasitism caused by parasitoids such as *Trichogramma*.



An egg tray made using 3 mm thick craft wood.

Calculating the levels of egg parasitism

The levels of parasitism can be estimated 2-3 days after you have collected the eggs. This is when all of the unparasitised eggs should have hatched, and you will see a small caterpillar in the cell of your egg tray. The number of eggs that don't hatch can be used to estimate parasitism, i.e.

$$\% \text{ Estimated egg parasitism} = (\text{UHE} / \text{TE}) \times 100$$

where:

UHE = The number of unhatched eggs in the egg tray.

TE = The total number of eggs in the egg tray.

The actual level of egg parasitism can be calculated 4-5 days after you have collected the eggs. This is when the parasitised eggs should have turned jet black.

$$\% \text{ Actual egg parasitism} = \text{BE} / (\text{BE} + \text{HE}) \times 100$$

where:

BE = The number of jet black parasitised eggs in the egg tray.

HE = The number of hatched eggs (caterpillars) in the egg tray.



Nymph and adult damsel bugs use piercing sucking 'beaks' to feed on many insects including *Helicoverpa* spp. eggs, larvae and mites.

Sometimes brown eggs collapse or don't hatch for other reasons and these unviable eggs should be excluded when you calculate the actual levels of egg parasitism, i.e. only use viable eggs. If you collect 60 brown eggs and you find that 45 turn jet black, 5 hatch and 10 collapse, then the actual level of egg parasitism is:

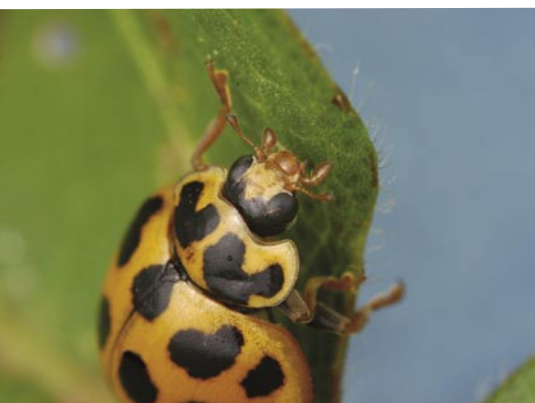
$$\begin{aligned} \% \text{ Actual egg parasitism} &= (45 / (45 + 5)) \times 100 \\ &= (45 / 50) \times 100 \\ &= 90\% \text{ egg parasitism} \end{aligned}$$

If you cannot collect eggs to assess *Trichogramma* activity, then you should look very carefully at the *Helicoverpa* counts. In particular you should look at the ratio of very small / small larvae to eggs. If *Trichogramma* are active you will find eggs, but few VS / S larvae. This indicates high egg, and / or early larval, mortality. However if you find as many VS / S larvae as eggs, then you know that egg hatch and early larval survival is high, which indicates low *Trichogramma* activity.

For more information on parasitoids of *Helicoverpa* spp., please contact: Entomologist, Agency for food and fibre sciences, DPI&F, Toowoomba, Queensland 07 4688 1200.

3.3.5 Use of beneficial insect attractants (food sprays) to conserve and enhance beneficial insects

The application of food sprays in cotton crops enables beneficial insects (particularly predators) to be attracted, retained and conserved in the cotton crop. Food sprays alone cannot manage cotton pests to achieve economically viable yields, although combined with other IPM compatible tools they can enhance the abundance of beneficial insects which will contribute to the control of pests and minimise the use of insecticides, without sacrificing yield. Commercially, there are two groups of food sprays made up of four different food spray products. The food spray groups are (1) yeast based food sprays and (2) sugar based food sprays. The yeast based food sprays are sold commercially as Envirofeed® and Predfeed® and the sugar based food sprays are Mobait® and Aminofeed®. The yeast based food sprays attract beneficial insects, whereas the sugar based ones retain them. Consequently these two groups are used differently to maximise their effectiveness. The yeast based food sprays should be applied when a cotton field does not have enough beneficial insects and there is the need to attract more into the field. In contrast, the sugar based food sprays should be used to maintain the beneficial insects already present in the field.



A common predator - the three-banded ladybird adult.

The guidelines described here make use of a predator / beneficial to pest ratio to incorporate the activity of the beneficial insects into pest management decisions. For the success of this type of IPM, it is important to select a field or whole farm that is not, or less likely, to be affected by insecticide drift as this will reduce beneficial abundance and reduce the likely success of this approach.

3.3.5.1 Decision making protocol for food sprays on conventional crops

The accepted beneficial to pest threshold is 0.5 or higher. Reference is made below to pest thresholds for *Helicoverpa*. These can be found in the latest 'Cotton Pest Management Guide' or in the section 'Pest thresholds' in objective 2.

- When the beneficial to pest ratio is 0.5 or higher and *Helicoverpa* numbers are below a threshold of 2 larvae per metre or the pest threshold is not exceeded, it means the IPM system is functioning well.
- When the ratio falls below 0.5 but is higher than 0.4 and *Helicoverpa* numbers are below threshold and the population is mostly eggs, a yeast based food spray can be applied to attract beneficial insects into the crop to feed on the eggs.
- If the beneficial to pest ratio falls below 0.5 but is higher than 0.4 and *Helicoverpa* numbers are below threshold but the population is predominately larvae (rather than eggs), then a sugar based food spray and biological pesticide or a petroleum spray oil (PSO) (Canopy® or Biopest oils® under permit) should be applied to restore the beneficial to pest ratio to 0.5 or higher by attracting predators and reducing *Helicoverpa* numbers.
- If a grower has lucerne strips or a centrally located lucerne crop on the farm, then before applying a food spray / biological insecticide spray, check the lucerne strip or crop to determine numbers of predators and adult green mirids. If beneficial insect numbers are high in the lucerne strips compared to cotton and numbers of adult mirids in the lucerne strips are low (< 5 per 20 metre d-vac sample), then slash half of each of the individual lucerne strips after applying the food spray / biological insecticide mixture. This action will enhance the movement of a large number of predators from the lucerne strips into the cotton, but will retain the mirids in the lucerne.
- In contrast, if both predator and adult mirid numbers in the lucerne strips are high (> 5 mirids per 20 metres), do not slash or mow the lucerne strips after applying the sugar based food spray / biological insecticide mixture since this will force too many mirids into the cotton where they may cause damage.
- If *Helicoverpa* larvae levels are above threshold in the next check following the application of a food spray / biological insecticide spray and the beneficial to pest ratio is 0.4 or lower, use a selective insecticide to control the larvae before they develop to mediums.



The beneficial to pest ratio does not include medium or large larvae as many predators prefer eggs or smaller larvae.

Table 9. Food spray decision making table for conventional and Bollgard II® cotton

Ratio	<i>Helicoverpa</i> spp.	Action
≥0.5	<2	Do nothing
0.4-0.5	<threshold (mostly eggs)	Yeast based food spray might be applied.
0.4-0.5	<threshold (mostly larvae)	Sugar based food spray, Biological pesticide or Petroleum spray oil (see section on lucerne below)
≤0.4	>threshold	Selective pesticide

3.3.5.2 Decision making protocol for food sprays on Bollgard II® crops

The application of the beneficial to pest threshold of 0.5 is essentially the same in Bollgard II® cotton as in conventional cotton. i.e. predators per metre / (*Helicoverpa* eggs + larvae (VS + S)) = 0.5 and above

- When the ratio is 0.5 or higher it means the IPM program on Bollgard II® is functioning well. Do nothing.
- If the ratio falls below 0.5 but is higher than 0.4 and the *Helicoverpa* population is below threshold and consists mostly of eggs (rather than neonates), apply a yeast based food spray to attract beneficial insects into the crop to feed on the eggs. This will help to restore the ratio to above 0.5.
- If the ratio falls below 0.5 but is higher than 0.4 and the *Helicoverpa* population is below threshold and consists mostly of neonates (rather than eggs), then a mixture of a sugar based food spray or a PSO (Canopy® or Biopest oils® under permit) as a stand alone or in mixtures with a biological insecticide should be applied. NB: The decision to slash half of the lucerne strips after food or PSO / biological insecticide application on Bollgard II® cotton should follow the same guidelines recommended for IPM on conventional cotton.
- If *Helicoverpa* neonate numbers in the next check following the application of a food spray or PSO / biological insecticide are above threshold but the check indicates an improvement in the beneficial to pest ratio, then re-apply a food spray / biological insecticide mixture to reduce *Helicoverpa* and help restore the ratio to 0.5 or higher. However if *Helicoverpa* neonate numbers in the next check following PSO / biological insecticide spray are above threshold and the check indicates no effect on the beneficial to pest ratio or the ratio is between 0.42-0.45 and larval numbers are still high, repeat the treatment.
- However, if the ratio has continued to decline after applying a food spray or PSO / biological insecticide mixture, reaching 0.4 or lower, and *Helicoverpa* are over threshold, then it is necessary to intervene with a selective insecticide to reduce *Helicoverpa* numbers before they moult to mediums.

3.3.5.3 The application of food sprays

Recommended rates of food sprays

Envirofeast® and Predfeed® products should be applied at 2.0 kg per hectare or label rate. Aminofeed® should be applied at 1 L / ha and Mobait® at 1 L / ha. Avoid applying the Aminofeed® attractant rate of 3 L / ha as studies have indicated it attracts *Helicoverpa* moths which may result in increased egg lays. Food spray rates can be applied as a band or skip row early season or over the entire field in the mid and late season. The first food spray of the season should be applied when the crop has 4-6 true leaves or when the number of *Helicoverpa* eggs reaches 1 per metre. Food sprays can be applied by ground or air as described below.

- *Ground rig application:* Application volume will vary according to the stage of growth of the cotton crop. Application to run off is recommended to ensure good coverage. The volume of food sprays may range from 80-120 L / ha depending on crop stage.
- *Aerial application:* A minimum application volume of 30-50 L / ha should be used.

Mixing and application of food sprays

Yeast based food sprays (Envirofeast® and Pred feed®) should be mixed as a slurry using a premix tank with agitation provided by a pipe connected to a water pump. Top up the mixture with water to reach the total volume required. The mixed product should be agitated constantly throughout mixing and transferral to the ground rig or aircraft.

Sugar based food sprays (Amino feed® and Mobait®) should be mixed by

adding the required rate of the product to the required volume of water or product solution.

All food sprays should be applied using flat fan nozzles or any nozzle that can concentrate the product on the top of the leaves. Coverage of the lower surface of the leaves is not essential, though an additional benefit will be gained from the product if the lower surface of the leaves are also covered.

To determine whether you have got the right concentration of yeast based food sprays on the leaves, look at them after the product has dried to see whether the leaf surface looks muddy. Also if the odour of the yeast is scented in the field this indicates there are residues of the product on the leaf. For sugar based food sprays look at the leaves to see whether the leaves look like a cream has been rubbed on them or are shiny compared to unsprayed plants.

Use food sprays the same day you mix them. With the yeast based food sprays, the product will go off or perish if the mixture or solution is left overnight. Clean ground rigs, aeroplanes, and premix tanks thoroughly with water before and after food spray to avoid fungal growth. Contamination of food sprays with the residues of earlier broad spectrum insecticides left over in spray tanks, may dramatically reduce predator numbers when applied to the cotton crop.

3.3.6 Using lucerne to manage mirids and as a nursery for beneficials

Cotton systems are moving toward less early season insecticide use due to the incorporation of Bollgard II® cotton and IPM. In the past, some pests, such as mirids, were often controlled unintentionally by early season insecticides applied for pests such as *Helicoverpa*. Hence mirids may become a more significant pest in the future and greater emphasis should be placed on managing them in IPM compatible ways. An option for non-chemical management of mirids is to use lucerne as a trap crop, as lucerne is more attractive to mirids than cotton.

For this option to work, lucerne needs to occupy a minimum of 2.5% of the whole cotton crop. It can be planted as strips consisting of 8, 12 or 16 rows of lucerne planted every 300 rows of cotton. However research has indicated that the preferred strategy is to plant lucerne as a block in a field adjacent to the cotton fields or as a centrally located block on the farm. The lucerne should be watered as needed to maintain active growth and half of each strip should be slashed or mowed every 4 weeks or as the lucerne begins to flower to maintain new growth and attractiveness to green mirids throughout the season. Mowing the entire strip will force the mirids into the cotton, as will letting the lucerne become stressed and unattractive.

The lucerne should be sown in mid to late autumn so it is established by the time the cotton crop emerges. In such a system it should not be necessary to use insecticides for the control of green mirids, however if insecticides are necessary, preference for more selective insecticides (low impact on beneficial insects) is essential.

The lucerne also serves as a nursery for beneficial insects. This is important because cotton fields are often large (50 - 150 ha) which means that re-colonisation of fields by beneficials may be slow after broad spectrum insecticide has been applied, especially if sources of predators are limited to farm perimeters. This approach can be further enhanced by use of a food spray to attract predators from the lucerne into the cotton and retain them there, enabling a degree of manipulation of predator / beneficial to pest ratios.

Size and placement of lucerne strips

Options for planting lucerne:

1. Strips within the cotton field at a ratio of 8, 12 or 16 rows of lucerne per 300 rows of cotton (i.e. lucerne area = 2.5% of whole field)



Slashing 1/2 of the lucerne every 4 weeks ensures that some is always attractive to mirids.

2. As a field border. It is preferable to plant lucerne on both sides of the field. In this case, a minimum area of 5% of the whole field should be planted to lucerne (i.e. 24 rows each side of a 1000 row wide field).
3. As a block. A lucerne crop can also be planted in a centrally located block on the farm.

Note: Options 2 and 3 may be slightly less effective than strips grown within the field for green mirid control. Also note that it is essential that the lucerne should remain green and not allowed to become dry or hay off.



A strip of lucerne grown between cotton.

Establishment

It is critical that the lucerne strips are established before cotton planting.

- *Sowing rate:* For lucerne strips to act as a trap crop, an effective plant stand needs to be established. Seeding rates of around 5 kg / ha should be used in dryland situations and 10 to 15 kg / ha for irrigated crops.
- *Seed bed preparation:* Good seed bed preparation is essential to achieve good establishment. For best results seek advice from an experienced lucerne grower.

Planting windows

Sowing time will vary according to the variety of lucerne selected and the growing district. In general, autumn and spring are the best periods for sowing. Avoid winter sowings in colder, wetter locations especially if the variety is winter dormant. Spring sowings should be used only under irrigation or in districts where spring rainfall is reliable.

Two planting windows have been used successfully:

- *April / June planting:* This produces a well established lucerne stand by the time the cotton is planted.
- *July / August planting:* Ensure lucerne is established by the end of August. This is the best option for establishing lucerne in back-to-back cotton fields. For irrigated crops, the lucerne strips should be formed into two equal beds by joining 4 rows into a bed in the case of an 8 row strip and 6 rows per bed in a 12 row strip. Run a furrow from head ditch to tail drain between the beds in each strip. This will ensure that water runs through the lucerne strip from head to tail during irrigation. However, if the lucerne is to be planted directly into the cotton beds, avoid planting in the furrows as this will block the furrow and create irrigation problems once the crop is established.

Irrigation management

For maximum production of lucerne in Northern NSW, irrigation should occur on a 20 day cycle from September to December and on a 10 day cycle from December onwards. Since cotton growers are not striving for maximum production from the lucerne the irrigation recommendations can be simplified to make them more compatible with typical cotton irrigation cycles. A first watering should be done when the cotton starts to square. Thereafter, to simplify irrigation management, irrigate the lucerne and the cotton at the same time. During the late cotton season, when broad spectrum insecticides such as synthetic pyrethroids are used on the farm or neighbouring farms, the lucerne is of no further use as a refuge or trap crop and can be slashed or mowed. Growers should note that a good stand of lucerne can contribute up to 200 kg N / ha in the soil every year in a crop rotation.

Management of mirids in lucerne strips or blocks

Green mirids prefer lucerne to cotton. Therefore, inter-planting lucerne strips in commercial cotton can be used to manage this pest. Lucerne grown for green mirid management should not be allowed to hay off. New regrowth of the lucerne should be maintained through the season by slashing or mowing half of each lucerne strip every 4 weeks, or when the crop is in flower as described below.

- Poorly established lucerne strips can reduce the crops ability to attract green mirids from cotton.
- It is important to do mirid counts and fruit retention counts in cotton

to confirm the level of mirid abundance (or damage) before applying a mirid spray. If an insecticide is applied to control green mirids select one that will have minimum impact on beneficial insects.

- The lucerne strips should be kept attractive through the cotton growing season, especially during the cotton crop's early squaring and flowering period. Once lucerne begins to flower, vegetative growth is limited and it is less attractive to green mirids. These could then move into cotton and cause problems. The attractiveness of the lucerne can be maintained by slashing or mowing half of each lucerne strip. This will ensure that the strip is always composed of older and younger lucerne growth. Mowing or slashing only half at a time means that the other half is still attractive to mirids. The first cut should be early, i.e. at or just before first square. Subsequent cuts should occur just as the other half starts to flower (provided there is sufficient re-growth in the other strips). Sometimes slashing may also occur in order to manipulate predator numbers in cotton.

Removing lucerne strips or blocks

Established lucerne can be killed either with cultivation or with herbicides. When the soil is dry, heavy cultivation such as a crawler with a cutter bar across the rippers has proved to be effective in removing established lucerne plants. The success of this technique requires dry soil during the cultivation and dry weather afterwards to prevent the lucerne from re-establishing. Herbicides are only effective for controlling lucerne when it is actively growing. Trial work shows that a tank mix of 2,4-D amine at 3 L / ha plus glyphosate at 1 L / ha will effectively control established lucerne, and most other broadleaf weeds present in the lucerne strip. However 2,4-D has a high risk of damaging cotton and extreme care should be taken with its application in cotton growing areas. The application should be made well before cotton planting as a 14 day plant-back period applies for cotton planted after herbicide application. This 14 day period only commences following rainfall of at least 15 mm. Thorough decontamination of spraying equipment is essential after 2,4-D applications.

For optimal control of lucerne, plants should be at least 10-15 cm tall and growing actively at the time of herbicide application. If moisture limitations are present and lucerne must be removed, cultivation is likely to give better control than herbicide options.

For further details on establishing and managing lucerne see DPI, NSW AGFACT P2.2.25 "*Lucerne for pasture and fodder*".

3.3.7 Conserving and enhancing parasitoids

There are some simple practices that can be employed to conserve and enhance parasitoids on a cotton farm, including:

- Not spraying insecticides of any kind unless it is necessary. This is best achieved by adhering to the general IPM guidelines and following the recommended thresholds for spraying, i.e. avoid spraying below threshold populations of pests.
- Choosing insecticides carefully when you have to spray. Some insecticides, such as Dipel® and Gemstar®, have very little impact on parasitoids. These and other selective products should be used to manage pests if possible. Broad spectrum insecticides, including the synthetic pyrethroids, are usually very toxic to parasitoids and should be avoided whenever possible.
- Maintaining habitat diversity on-farm. This can be achieved by growing a mixture of crops and avoiding cotton monocultures. Sorghum, maize and sunflowers are all good nursery crops for parasitoids. Sunflower is a nursery crop for green mirids and can become a source of green mirid infestation in the cotton crops. Avoid using sunflower as a parasitoid nursery unless there is a lucerne crop adjacent to the cotton. The capacity of sorghum and maize to act as parasitoid nurseries can be



Sorghum can be an effective nursery crop for parasitoids.

extended by growing staggered plantings, i.e. by sowing these crops on 2 or more separate occasions. Some crops, such as chickpea, are not good nursery crops for all parasitoids i.e. *Trichogramma* are not effective in chickpea because the acidic chickpea leaves are toxic to the adult wasps. It is important to manage pests carefully in the nursery crops to conserve parasitoids. For example, if you have to spray sorghum to manage *Helicoverpa* try to use Gemstar®, or another selective insecticide, so that you don't kill the *Trichogramma*, *Microplitis* and other parasitoids that may be in the crop. Refer to Table 10, 'The impact of insecticides and miticides on predators in cotton'.

Bollgard II® cotton will act as a nursery for parasitoids because it will receive few insecticide sprays. If you find high levels of egg parasitism in a Bollgard II® crop then manage the non *Helicoverpa* pests carefully. By conserving the parasitoids they may move into adjacent, or nearby, conventional cotton and other crops.

3.3.7.1 Releasing *Trichogramma*

The *Trichogramma* wasp is an egg parasitoid which is a very important beneficial causing high mortality of *Helicoverpa* spp. The wasp kills its host by laying an egg inside a *Helicoverpa* egg. The wasp larvae then feeds on the developing *Helicoverpa* larvae killing it before it hatches.

Trichogramma can be purchased and released into crops to kill *Helicoverpa* eggs. They are available from 'Bugs for Bugs', Mundubbera Queensland, and the company should be contacted for details of costs and release methods.

One release method that is suitable for small acreages is the use of small cardboard capsules. Each capsule contains about 1,000 wasps, and they can be stapled to cotton leaves to prevent them from falling on the ground. This prevents the possible exposure of the wasps to potentially deadly high soil temperatures.

It is important not to think of *Trichogramma* releases as a substitute for an insecticide spray. They will give inconsistent results if they are used in this fashion. This is primarily because *Helicoverpa* eggs hatch in 2-3 days during summer, and many may hatch before you can order *Trichogramma* and release them into your crop.

It is best to think of *Trichogramma* as part of an IPM program, where the careful selection of 'soft' insecticides can be used in conjunction with the wasps to manage *Helicoverpa*. In recent years some cotton growers have released wasps to 'kick start' *Trichogramma* populations, i.e. to establish populations before they would naturally appear. This approach, called inoculative release, normally involves completing two releases of 30 capsules per hectare about a week apart. This process can be completed in cotton, or in adjacent crops on your farm, e.g. sorghum or maize. Avoid spraying the release crops with broad spectrum insecticides so that you don't kill the *Trichogramma*. You may not notice an immediate impact from the inoculative releases, but you should notice an impact after the wasps have completed one generation in the field, i.e. about 10 days after a release.

A key difference between predators and *Trichogramma* is the nature of their impact. An egg that is eaten is removed from the crop and isn't counted by consultants. However, an egg that is parasitised remains in the crop and can be accidentally counted by consultants, unless it has turned jet black and is recognised as being parasitised. So you really need to assess egg parasitism if you want to avoid unnecessary sprays, and get the most out of your *Trichogramma*. For more information on *Trichogramma* parasitism refer to 'Sampling and determination of *Trichogramma* parasitism' in this objective.



A *Trichogramma* wasp emerging from a parasitised *Helicoverpa* egg.

3.3.8 *Tolerate non-economic early season damage to help conserve beneficials*

The cotton plant has the ability to compensate for a reasonable level of damage without affecting yield or crop maturity. It is therefore important to monitor leaf and tip damage on pre-squaring cotton and to assess fruit retained early in the season (post-squaring to flowering) and to tolerate a level of non-economic damage (refer to Table 8, '*Pest threshold summary*' in objective 2). Combining pest and damage thresholds to assist with pest management decisions, maximises the opportunity to reduce the number of insecticide applications which consequently helps to conserve the beneficial insect population (Refer to section '*Early season damage*' in objective 2).

3.3.9 *Insect resistant transgenic cotton varieties*

Bollgard II® cotton is ideally suited to IPM as the level of control of *Helicoverpa* spp. provided by the plant is usually sufficient to dramatically reduce the need to spray for this pest or other lepidopteran pests such as tipworm, especially early season. This provides an opportunity to retain a good population of beneficial insects in the crop. However, Bollgard II® cotton does not provide protection against pests such as mirids, so it is important to monitor the insects and damage in the same way as a conventional crop.

3.3.10 *Appropriate use of insecticides*

The use of insecticides is often the major factor limiting the buildup of beneficial insects on cotton farms. When using insecticides there are some simple practices that can be employed to conserve and enhance beneficial insects on your farm. They include:

- Not spraying insecticides of any kind unless it is necessary. This is best achieved by adhering to the general IPM guidelines and following the recommended threshold for spraying, i.e. avoid spraying below threshold populations of pests (refer to Table 8, '*Pest threshold summary*' in objective 2).
- Choosing insecticides carefully when you have to spray. Some insecticides, such as Dipel® and Gemstar®, have very little impact on beneficial insects including parasitoids. These and other selective products should be used to manage pests, if possible. Broad spectrum insecticides including the synthetic pyrethroids are usually very toxic to beneficial insects and should be avoided whenever possible. Refer to Table 10, '*The impact of insecticides and miticides on predators in cotton*'.
- Site specific pest management. Many beneficial insects frequently move in and out of cotton, other crops and non-crop habitats. Therefore the timing and location of insecticide applications must be carefully considered. It is important to manage pests on a field by field basis or by a small management unit, not an entire farm or large management unit. This way, only those fields that actually require pest control should be sprayed. This leaves other fields which are not being sprayed to serve as sources of beneficials to help re-colonize sprayed fields. Pests such as aphids or mites often infest the edge of a field, not the entire field area. It is therefore possible to manage this type of infestation by only spraying the field borders. This minimises the field area treated which is likely to kill a smaller proportion of beneficials and enables the beneficial population to re-establish much faster.



Checking Bollgard II® cotton for pests and beneficials.

Table 10. The impact of insecticides and miticides on beneficial insects in cotton

Insecticides (in increasing rank order of impact on beneficials)	rate (g ai / ha)	Target Pest(s)					Beneficials															Pest resurgence ¹²		Toxicity to bees ¹⁴			
		Helicoverpa	Mites	Mirids	Aphids	Thrips	Persistence ³	Overall ranking ¹⁰	Predatory beetles				Predatory bugs				Lacewing adults	Spiders	Hymenoptera			Thrips	Mite		Aphid	Helicoverpa	
									Total ¹	Red & Blue beetle	Minute 2-sp t lady	Other Lady Beetles	Total ²	Damsel bugs	Big-eyed bugs	Other Predatory			Apple Dimpling	Total (wasps)	Trichogramma						Ants
Bt ¹¹		•				very short	very low	VL	VL	VL	VL	VL	VL	VL	VL	VL	VL	VL	VL	VL	VL				VL		
NP Virus		•				very short	very low	VL	VL	VL	VL	VL	VL	VL	VL	VL	VL	VL	VL	VL	VL				VL		
Pirimicarb	250				•	short	very low	VL	VL	VL	VL	L	L	M	VL	VL	VL	VL	VL	H	VL	L			VL		
Methoxyfenozide	400	•				medium long	very low	L	VL	VL	L	L	L	L	VL	VL	VL	VL	VL	VL	VL	VL			VL		
Dicofol ²	960		•			long	low	L				L				L		L				VL			VL		
Spinosad	96	•				medium	low	VL	M	L	VL	M	L	H	VL	L	VL	VL	M	H	H	H	+ve		H ¹⁵		
Diafenthiuron	350		•			medium	low	M	H	VL	M	L	M	VL	L	H	VL	L	L	L	H	L			+ve	M	
Pymetrozine	150				•	short	low	M	M	M	M	L	L	L	VL	H	M	L	L	L	M	VL			VL		
Indoxacarb ¹³	127.5	•		•		medium	low	H ¹⁴	L	VH	VH	L	L	L	L	VH	L	VL	L	L	VH	VL		+ve	H ¹⁵		
Abamectin	5.4	+6	•			medium	moderate	L	M	H	VL	M	L	M	M	H	VL	M	M	M	H	M			H		
Emamectin	8.4	•				medium	moderate	L	VL	M	VL	H	H	H	H	H	L	M	M	M	VL	M			H		
Endosulfan (low)	367.5	•		•	•	•	medium	moderate	M	VL	VH	M	M	M	M	L	H	L	M	VL		VH	H		M ¹⁵		
Propargite	1500		•			medium	moderate	M	H	H	M	M	M	H	VL	VL	L	VL	M	M	H	H	M	+ve	+ve	L	
Acetamiprid	22.5				•	medium	moderate	M	M	VH	H	M	M	H	M	VH	L	VL	L	VH	VH	VH			M ¹⁵		
Amitraz	400	•	+9			medium	moderate	H	M	VH	H	L				H	VL	M	M	M	H	M			L		
Fipronil (low)	12.5			•		•	medium	moderate	L	VL	H	L	L	H	L	L	VH	VL	M	M	VH	VH	VH	+ve	+ve	VH	
Chlorfenapyr (low)	200	•	•			medium	moderate	M	L	VH	VL	M	VL	H	H	VH	L	L	M	VH	H	M			H		
Thiamethoxam	100				•	medium	moderate	H	H	H	H	M	M	M	M	H	H	M	VL	M	H	VH	H	+ve	+ve	H	
Endosulfan (high)	735	•		•	•	•	medium	moderate	M	VL	VH	M	M	M	M	M	H	L	M	L	VH	VH	H		M ¹⁵		
Fipronil (high)	25			•		•	medium	moderate	L	VL	H	L	M	H	H	L	VH	VL	M	M	VH	VH	VH	+ve	+ve	VH	
Imidacloprid	49			•	•		medium	moderate	H	L	VH	H	H	M	H	L	VH	L	L	L	VH	H	H	+ve	+ve	M	
Methomyl	169	•				very short	high	H	L	VH	VH	M	L	VH	L	VH	M	M	M	M	H	H	H	+ve		H ¹⁵	
Thiodicarb	750	•				long	high	VH	M	VH	VH	M	M	L	L	VH	VL	M	M	H	M	H		+ve	M ¹⁵		
Chlorfenapyr (high)	400	•	•			medium	high	H	M	VH	L	H	H	H	H	VH	L	M	M	VH	VH	M		+ve	H		
OP's ⁵		•	•	•	•	•	short-medium	high	H	M	H	H	H	M	H	H	VH	L	M	H	H	VH	H	+ve		H	
Carbaryl ³						short	high	H					H					-	-	-	-	H			H		
Pyrethroids 4		•	+7	+7		long	very high	VH				VH					VH	VH	VH	VH	VH	VH	VH	+ve	+ve	+ve	H

1. Total predatory beetles – lady beetles, red and blue beetles, other predatory beetles
2. Total predatory bugs – big-eyed bugs, minute pirate bugs, brown smudge bugs, glossy shield bug, predatory shield bug, damsel bug, assassin bug, apple dimpling bug
3. Information; Citrus pests and their natural enemies, edited by Dan Smith; University of California Statewide IPM project, Cotton, Selectivity and persistence of key cotton insecticides and miticides.
4. Pyrethroids; alpha-cypermethrin, cypermethrin, beta-cyfluthrin, cyfluthrin, bifenthrin, fenvalerate, esfenvalerate, deltamethrin, lambda-cyhalothrin,
5. Organophosphates; dimethoate, omethoate, monocrotophos, profenofos, chlorpyrifos, chlorpyrifos-methyl, azinophos ethyl, methidathion, parathion-methyl, thiometon
6. *Helicoverpa punctigera* only.
7. Bifenthrin is registered for mite control; alpha-cypermethrin, beta-cyfluthrin, bifenthrin, deltamethrin and lambda-cyhalothrin are registered for control of mirids
8. Persistence of pest control; short, less than 3 days; medium, 3-7 days, long, greater than 10 days.
9. Suppression of mites only.

10. Impact rating (% reduction in beneficials following application, based on scores for the major beneficial groups); VL (very low), less than 10%; L (low), 10-20%; M (moderate), 20-40%; H (high), 40-60%; VH (very high), > 60%. A '-' indicates no data available for specific local species.

11. *Bacillus thuringiensis*
12. Pest resurgence is +ve if repeated applications of a particular product are likely to increase the risk of pest outbreaks or resurgence. Similarly sequential applications of products with a high pest resurgence rating will increase the risk of outbreaks or resurgence of the particular pest species.
13. Very high impact on minute two-spotted lady beetle and other lady beetles for wet spray, moderate impact for dried spray.
14. Data Source: British Crop Protection Council. 2003. The Pesticide Manual: A World Compendium (Thirteenth Edition),. Where LD50 data is not available impacts are based on comments and descriptions. Where LD50 data is available impacts are based on the following scale: very low = LD50 (48h) > 100 ug / bee, low = LD50 (48h) < 100 ug / bee, moderate = LD50 (48h) < 10 ug / bee, high = LD50 (48h) < 1 ug / bee, very high = LD50 (48h) < 0.1 ug / bee. Refer to the Protecting Bees section in this booklet.
15. Wet residue of these products is toxic to bees, however, applying the products in the early evening when bees are not foraging will allow spray to dry, reducing risk to bees the following day.

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3.3.11.1 *The beneficial disruption index*

The Beneficial Disruption Index (BDI) provides a basis to measure or benchmark the ‘softness’ or ‘hardness’ of an individual fields’ insecticide spray regime at the end of the season. The BDI score for each insecticide is based on the overall impact of the insecticide on beneficial insect populations, as listed in Table 10. A chemical that is more disruptive has a higher score or rank. Each BDI point equals a 10% reduction in beneficials after application of the chemical. The overall BDI for a cotton field is calculated by summing all the BDI scores for each insecticide used over the whole season. Note that scores for each component of spray mixtures are added together. The lower the overall rank for the season the more friendly the spray regime.

BDI point	Impact rating from Table 10.
1	VL (very low), less than 10%
2	L (low), 10-20%
4	M (moderate), 20-40%
6	H (high), 40-60%
8	VH (very high), > 60%



3.4 Objective 4 - Prevent the development of insecticide resistance

3.4.1 Introduction

Resistance occurs when application of insecticides removes susceptible insects from a population leaving those individuals that are resistant. Mating between these resistant individuals gradually increases the proportion of resistance in the pest population as a whole. Eventually this can render an insecticide ineffective, leading to field control failures. Resistance can be due to a trait that is already present in a small portion of the pest population or due to a mutation that provides resistance.

Management of resistance is essential to ensure that valuable insecticides remain effective. One of the objectives of IPM is to help manage insecticide resistance by reducing the overall use of insecticides, which reduces the number of selection events. However IPM programs rely on the availability of selective insecticides i.e. those that control only the target pest with little effect on beneficials as an important tool to manage pests. IPM and resistance management are therefore complimentary.

To help manage resistance the Australian cotton industry has developed the Insecticide Resistance Management Strategy (IRMS). This industry regulated strategy sets limits on which insecticides can be used, when they can be used and how many times they can be used. These limits are based on the outcomes of ongoing monitoring for resistance in *Helicoverpa* spp., mites, aphids and whiteflies, and are designed to prevent resistance developing or to manage resistance that has developed. The IRMS is usually devised for 2 or 4 major groups of regions, reflecting differences in pest pressure or timing of crop growth. For the success of the IRMS, it is important for every grower to follow their regional strategy. Most pests are not confined to a particular field or farm, so if one grower acts outside the strategy causing insecticide resistance on his / her farm, then it is possible for the resistant pest population to spread to neighbouring farms hindering the efforts of other growers who have been diligent in their pest management.

Resistance monitoring

Resistance monitoring for *Helicoverpa* spp., mites, aphids and whiteflies, is conducted each year and provides the foundation for annual review and updating of the strategy. The monitoring program is a coordinated effort between researchers, consultants, growers and extension staff. In addition to resistance monitoring, research is also carried out on the mechanisms of resistance. This is useful for devising strategies that avoid cross resistance and can also lead to field kits for identification of resistant insect populations before making spray decisions.

The IRMS is designed to prevent resistance development, while managing existing resistance. Limiting the number of applications permitted for new insecticides is an example of pro-active management. The IRMS is responsive to changes between seasons as new information becomes



Testing for
insecticide resistance
in the laboratory.

available from resistance monitoring. However, every season is different and the weather and insect patterns of the previous season should not overly influence the long term aim of resistance management.

Resistance mechanisms

The main mechanisms of resistance are given below. However, resistance is complex and there can be number of mechanisms present in a single pest, to one or more insecticides:

- *Target site insensitivity*: changes to the target site proteins reduce binding of the chemical and hence reduce its toxicity.
- *Metabolic resistance*: chemicals are broken down to non-toxic substances by increased enzyme activity in the insect before they can act on the target site. (e.g. this is the most common mechanism in *H. armigera* for detoxifying organophosphates, pyrethroids & carbamates).
- *Penetration resistance*: changes in the insect's cuticle or gut lining (if ingested) can lead to slower uptake of chemicals. Normal detoxification enzymes can then act.
- *Altered behaviour*: the target insect's behaviour changes in a way that may allow them to avoid normally lethal spray deposits. E.g. they may move down the plant away from the spray or cease feeding on sprayed foliage.
- *Cross resistance*: the resistance situation is further complicated by resistance to one group of insecticides conferring resistance to another, referred to as cross resistance. An example of this is resistance in the cotton aphid to carbamates that also confers resistance to the organophosphates.

Resistance management tools

Resistance management relies on a number of core principles. The exact details may vary between pest species due to differences in life cycle, host range and ecology. Management of resistance to the Bt toxins (proteins produced in cotton by genes inserted from *Bacillus thuringiensis* var *kurstaki*) in Bollgard II® varieties and insecticides applied to the plant are essentially similar except that the options to rotate the toxin or restrict the number or timing of applications of the toxin are not possible with the transgenic plants. Some core principles used in the Australian IRMS include:

1. Limiting the time period during which an insecticide can be used. This restricts the number of generations that can be selected.
2. Limiting the number of applications, thereby restricting the number of selection events.
3. Rotation between chemical groups with different modes of action. Insecticides are grouped according to their mode of action (the way in which they kill the pest). There may be a range of insecticides in a particular group. Repeated use of insecticides from one chemical group can increase the selection pressure against that mode of action. If resistance develops it will usually affect all insecticides in the same group. Rotation between chemical groups reduces selection for a particular mechanism that is effective against a particular insecticide group. For fast breeding pests such as mites, aphids and whiteflies it is recommended that no insecticides / miticides from the same group are used consecutively. For *H. armigera*, up to two applications of the same group can be made, then rotation must occur.
4. Reducing overwinter survival of pests that have been selected for resistance. *H. armigera* can survive through winter as diapausing pupae in the soil. These pupae are in a state of arrested development through winter. When conditions warm up they will resume development. The trigger for pupae to go into diapause is reduced day length and temperature. Toward the end of the cotton season most pupae will go into diapause, and these will have been selected for resistance to insecticides, or to the Bt proteins in Bollgard II®. They can be controlled by cultivation thereby reducing the carryover of resistance from



Cultivating to control diapausing *H. armigera* pupae.

one season to the next. Mites, aphids and whiteflies all use weed or crop hosts during winter. Reducing the availability of these hosts will reduce the size of populations that infest cotton in the next season, and thereby reduce the number of insecticide applications and selection events.

5. Use of trap crops to concentrate a pest in a particular area where they can be controlled by other means i.e. destructive cultivation of the crop.

Several other strategies can also help in managing resistance. These include:

1. Selective insecticide use, consistent with the IRMS, helps conserve beneficial insects. Beneficials eat or parasitise resistant as well as susceptible pests. Beneficials can also lower overall populations of insect pests.
2. Using plant compensation allows for the plant's capacity to recover from a degree of damage without loss, thereby avoiding insecticide applications to prevent non-economic damage.
3. Avoid cross selection for resistance. Spraying for one pest can be simultaneously selecting resistance in another pest that is present, even though that pest is at sub-threshold levels.

3.4.2 Strategies for individual pests

Each pest has a different life cycle and ecology which means the strategy required to manage resistance may vary between species. Several examples for important pests are given below.

3.4.2.1 *Helicoverpa*

Helicoverpa spp. have a relatively long life cycle (42 days or about 5 generations in a season) which makes this pest ideally suited to a resistance management strategy based on restricting chemical use to defined time periods corresponding to its generation time.

Of the two *Helicoverpa* species in cotton, *H. armigera* has developed the greatest resistance. This species is closely linked with cropping systems and is exposed to insecticides on many of these hosts. *H. punctigera* has the biological capacity to become resistant, and there are populations with low levels, but its ecology involves frequent migration to and from untreated hosts which dilutes any resistance.

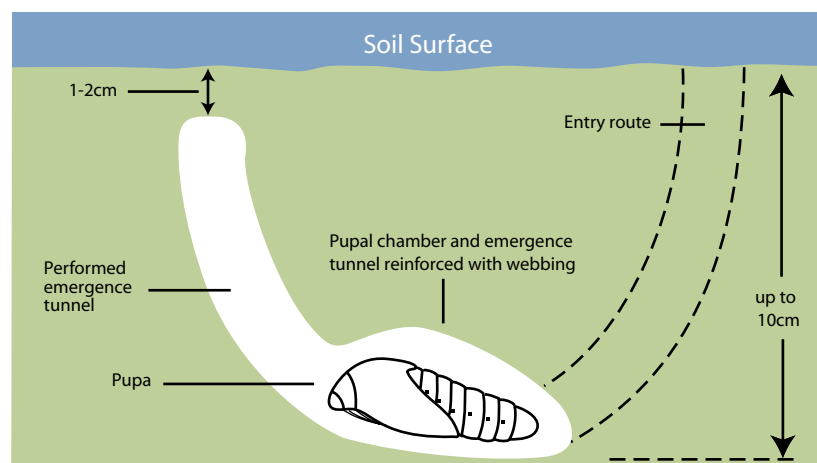
H. armigera are mobile pests which can move between cropping regions given favourable weather conditions. This makes it necessary to have an industry wide strategy that coordinates resistance management. The strategy also accounts for control of this pest in crops other than cotton, which is particularly important for insecticides used on more than one crop.

The pupal stage of the *Helicoverpa* life cycle normally lasts about two weeks. Older larvae leave the cotton plant and burrow into the soil. They form emergence tunnels, then turn into pupae. While in the pupal stage they undergo physiological changes into moths. The moth cannot dig so it uses the emergence tunnel to leave the soil (Figure 9).



Large *Helicoverpa* larvae will move down to the soil to pupate.

Figure 9.
Position of the *Helicoverpa* pupa in the soil with entry and emergence tunnels.
Illustration: J. Beard



In autumn and winter, short day lengths and cool temperatures can trigger a proportion of pupae to go into diapause. This is a dormant phase that allows them to survive in a state of suspended development for several months. When soil temperatures increase in spring, normal development is resumed and moths emerge soon afterwards.

Diapause generally commences at the end of the cotton season, when the levels of insecticide resistance in *H. armigera* are at their highest. Moths that emerge in the following spring from diapausing pupae are likely to be highly resistant. In fact, these individuals are the major carriers of resistance from one season to the next. Cultivation to control insecticide resistant *H. armigera* pupae under cotton stubble (pupae busting) is a core non-chemical component of both IRMS and IPM.

Sample cotton stubble for pupae after harvest, using the guidelines published, in order to determine which fields require control and to prioritise those that do. Cultivate to control pupae as soon as possible after harvest and no later than August. Early cultivation will also kill any non-diapausing pupae remaining in the soil. Early cultivation also increases the chance of later rain events sealing up any remaining pupae emergence tunnels, preventing moths from emerging. Minimum tillage strategies for planting rotation crops may lead to poor pupae control. *MACHINEpak* and the *Cotton Insect Pest Management Guide* provide information on the effectiveness of different cultivation options for controlling pupae. Avoid cultivating under conditions that create other problems such as compaction, i.e. wet soil.

The stubble of other summer crops may also harbour *Helicoverpa* pupae. Sample to assess pupae densities under these crops as soon as possible after harvest and pupae bust if warranted. Grain crops not infested with *Helicoverpa* larvae by early March will not harbour diapausing pupae unless there has been re-growth.

To find out whether your pupae busting has been effective visit www.cotton.pi.csiro.au/Assets/PDFFiles/evalpup.pdf

The proportion of pupae predicted to enter diapause on a given date for several cotton growing regions is given in Tables 11-15. These predictions are based on the model of diapause induction for *H. armigera* developed by Dr David Murray using field cages on the Darling Downs. The HEAPS (*Helicoverpa Armigera* and *Punctigera* Simulation) model has been used to estimate the time taken for eggs laid each week throughout February and March to develop to the pupal stage, and the proportion of insects that pupate on a given date that are likely to enter diapause. Pupae that do not enter diapause will continue their development, and their predicted emergence dates are also given.

Diapause induction is a complex process that is influenced by decreasing day length and daily temperature cycles. The model predictions given here should be considered as estimates only, because cotton fields in different locations within each region will experience different regimes of day length and temperature. There will also be seasonal variations. In cool seasons diapause induction may commence up to two weeks earlier, and in warm seasons diapause will occur later than average.

Contact your local industry development officer or district agronomist for the current induction data for your region.

The following tables are *H. armigera* autumn diapause induction and emergence dates of non-diapausing pupae for major regions (Central Qld and MacIntyre) based on long term average temperatures.



Helicoverpa pupae.



Sampling for diapausing *Helicoverpa* pupae under crops in autumn or winter is critical for deciding which fields need to be cultivated to control pupae.



The stubble of other crops such as sunflowers may also harbour *H. armigera* pupae. Check to see if cultivation is required.

Table 11. Central Queensland *H. armigera* autumn diapause induction and emergence dates

Date of egg lay	Pupation	% Diapause	Non-diapause emergence
1 February	21 February	0.0	5 March
8 February	25 February	0.0	10 March
15 February	7 March	2.1	19 March
22 February	12 March	15.7	25 March
1 March	22 March	42.0	5 April
8 March	29 March	57.8	15 April
15 March	6 April	74.7	23 April
22 March	15 April	90.9	18 May

Table 12. Macintyre *H. armigera* autumn diapause induction and emergence dates

Date of egg lay	Pupation	% Diapause	Non-diapause emergence
1 February	25 February	0.0	11 March
8 February	3 March	0.0	18 March
15 February	11 March	11.4	28 March
22 February	19 March	29.1	6 April
1 March	27 March	46.8	17 April
8 March	5 April	64.4	30 April
15 March	14 April	78.6	20 May
22 March	25 April	92.4	12 June

Table 13. Namoi *H. armigera* autumn diapause induction and emergence dates

Date of egg lay	Pupation	% Diapause	Non-diapause emergence
1 February	28 February	0.0	17 March
8 February	9 March	4.2	27 March
15 February	15 March	17.5	3 April
22 February	24 March	38.0	17 April
1 March	31 March	50.9	28 April
8 March	11 April	68.3	22 May
15 March	23 April	83.6	16 June
22 March	7 May	94.0	29 June

Table 14. Macquarie *H. armigera* autumn diapause induction and emergence dates

Date of egg lay	Pupation	% Diapause	Non-diapause emergence
1 February	23 February	0.0	13 March
8 February	3 March	0.0	25 March
15 February	14 March	17.6	6 April
22 February	21 March	31.9	13 April
1 March	2 April	51.0	7 May
8 March	11 April	68.8	25 May
15 March	1 May	79.5	23 June
22 March	12 May	91.1	4 July

Predicted emergence of H. armigera from diapause in spring

The proportion of diapausing pupae predicted to resume normal development and emerge as moths is given in Table 15 for each of the major cotton growing regions based on long term average temperatures. These predictions were made using the model of diapause termination for *H. armigera* developed by Dr David Murray. Diapause termination is influenced by soil temperatures. The model predictions should be considered as estimates only, because the interactions between soil temperature, moisture, and pupal depth will be strongly influenced by local conditions at the field

level. In cool seasons these emergence periods may be delayed by up to two weeks, and in warm seasons emergence will occur earlier than average. Emergence from diapause is a long process that generally takes place over 6 to 8 weeks.

The following table shows the predicted spring emergence of *H. armigera* moths from winter diapause. Contact your local industry development officer or district agronomist for the current emergence data for your region.

Table 15. Spring emergence of *H. armigera* moths

Region	1% emergence	50% emergence	99% emergence
Central QLD	14 August	3 September	6 October
Macintyre	28 September	23 October	23 November
Gwydir	1 October	26 October	25 November
Namoi	4 October	29 October	28 November
Macquarie	21 October	13 November	12 December
MIA	27 October	17 November	9 December
D. Downs	1 November	12 November	20 December

3.4.2.2 Fast life cycle pests (mites, aphids, whiteflies)

The current strategy window system was originally developed for *Helicoverpa* spp. as the major pest. However with the introduction of Bt cotton and an industry wide acceptance of IPM for pest management, other secondary pests that once would have been controlled inadvertently through *Helicoverpa* spp. control are now increasing in importance. Therefore the strategy has now incorporated resistance management plans for secondary pests.

Aphids, mites and whiteflies have very short life cycles, 5, 8 and 16 days respectively under summer conditions. This allows them to develop resistance to insecticides very quickly. It is important not to consecutively apply insecticides from the same group for control of these pests or when selecting chemistry for other pests. This strategy avoids selection of multiple generations with the same insecticide group. This applies even if short life cycle pests are below thresholds in pest checks.

Rotation between insecticide groups is especially critical with pests that reproduce by cloning such as cotton aphids (see the '*Cotton Pest and Beneficial Guide*', or the '*Managing Aphids Research Review*' available on the Australian Cotton CRC website). Any resistant survivors will pass their resistance trait directly to their offspring. In cotton aphid populations in Australian cotton regions there is no sexual reproduction and therefore no potential for dilution of resistance by mating with susceptible insects, as there is with most other pests.

Aphids, mites and whiteflies all use a wide range of hosts so management of these through winter to reduce the size of overwintering populations is critical in their management.

Aphid specific issues

For cotton aphids (*Aphis gossypii*) there is cross resistance between pirimicarb (carbamate), omethoate and dimethoate (organophosphates). Early season use of these chemicals risks re-selecting overwintering resistant aphids. The resistant strains are however susceptible to some of the other organophosphates and endosulfan, as well as neonicotinoids (imidacloprid (Confidor®), thiamethoxam (Actara®) and acetamiprid (Intruder®)), pymetrozine (Fulfil®) and diafenthiuron (Pegasus®).

To complicate matters, use of pyrethroids against *Helicoverpa* can exacerbate resistance problems by selecting the pirimicarb / organophosphate resistant aphids to create pirimicarb / organophosphate / pyrethroid resistant clones that are highly resistant to virtually all organophosphates, carbamates and pyrethroids making them very difficult to control, especially toward the end of the season.



Cotton aphids reproduce parthenogenically, so young are clones of their mother. This reduces the chance for dilution of resistance.



Aphid parasitoid (*Lisiphlebus testaceipes*).



Mites use weed and crop hosts (like this marshmallow) to survive through winter. Good farm hygiene can help reduce over-winter survival of mites, aphids and whiteflies.



Mites have a relatively high reproductive potential of about 70 eggs per female.



The silverleaf whitefly (SLW), *Bemisia tabaci*, B-biotype is resistant to most insecticides.

To avoid this problem, pirimicarb, omethoate and dimethoate need to be used less and more strategically. Pirimicarb's strategic use is with early season IPM as it is soft on many of the beneficial groups that control aphids. Omethoate and dimethoate's strategic use is at the end of the season where their negative impact on beneficials is less critical. This allows the softer chemistry to be used earlier, better utilising beneficial insects to reduce surviving pests.

If using a seed or in furrow treatment from the neonicotinoid group avoid using an insecticide from this group as your first foliar spray.

Cotton aphids can overwinter on numerous crops, weed hosts and cotton re-growth or volunteer cotton. There is a risk of resistant clones persisting on-farm through winter. It is therefore important to reduce the availability of host plants over the winter period through the control of winter weeds and careful choice of winter crops. On farms where aphid resistance occurred in the last season, the grower should consider planting a winter cereal or a winter legume that is not a good aphid host, such as retch.

For more information on aphid management visit www.cotton.pi.csiro.au/Assets/PDFFiles/AphMang.pdf

Mite specific issues

Most miticides are also used against other pests. It is critical to be aware of this issue as use against another pest also selects for resistance in mites present in the field, even if they are at sub-threshold levels. Two-spotted spider mites (*Tetranychus urticae*) are widely resistant to organophosphate insecticides, to bifenthrin (Talstar®) and increasingly to chlorfenapyr (Intrepid®).

Mites survive the winter in colonies on broadleaf weeds and crops such as safflower, faba bean and field pea. As these hosts senesce in spring, mites migrate to new hosts such as cotton seedlings. It is therefore important to practice good farm hygiene to control winter weeds and if you have experienced mite infestations in the past, carefully choose your rotation crop. Winter cereals and chickpeas are poor mite hosts.

Mite survival may also increase if the winter rotation crop is sprayed with a broad spectrum insecticide to control other pests, as this will reduce the population of mite predators.

In late maturing cotton crops, mites can enter diapause and move to the base of the plant into the cotton trash or cracks in the soil. Cultivation, for the purpose of pupae busting will help reduce the survival of these mites.

For more information on mite ecology visit www.cotton.pi.csiro.au/Assets/PDFFiles/miteeco.pdf.

Whitefly specific issues

The silverleaf whitefly (SLW), *Bemisia tabaci*, B-biotype is resistant to most insecticides used for control (including OPs, carbamates, pyrethroids, imidacloprid and insect growth regulators (IGRs)). Management of this pest hinges on an IPM strategy that incorporates biological, cultural & insecticidal management practices. This includes the avoidance of broad spectrum sprays and the strategic early use of IGRs to slow the growth of whitefly populations while allowing the buildup of beneficial insects to maintain whitefly at sub-economic levels. The IGR's are at risk due to resistance so use is limited to one application during a defined window within the season.

Although SLW has only been an issue in Central Queensland it has the potential given the right environmental and cropping conditions to be a problem elsewhere in the industry. Monitoring SLW populations is therefore conducted to provide an early warning of potential infestations.

The SLW does not have an overwintering diapause stage, and in warm areas can survive through the winter. A major factor to reduce the likelihood of a SLW outbreak is to discontinue the availability of host plants. This occurs in regions such as northern NSW and the Darling Downs where the predominant rotation is cotton / cereal or cotton / legume which are not particularly

good hosts for the SLW. In regions such as the Emerald irrigation area, the continuous availability of suitable hosts such as horticultural vegetables, particularly cucurbits, encourages SLW outbreaks. Weed cucurbits or thistles should be controlled as they can also act as reservoirs for whitefly to survive through the winter months.

For further information on managing SLW visit www.cotton.pi.csiro.au/Assets/PDFFiles/Wflymng.pdf

3.4.2.3 Other pests can develop resistance

The sucking pests (mirids and green vegetable bugs) previously controlled by broad spectrum sprays for *Helicoverpa* are becoming more common, particularly as greater areas of Bollgard II® cotton are planted with the associated reduction in spray numbers.

Although the resistance management strategy does not specifically mention these pests, they may develop resistance. It is important to consider this when controlling sucking pests as the over reliance on any one insecticide or insecticide group increases the risk of selecting for resistance.

Winter weeds and crops such as safflower are good winter hosts for mirids. It is therefore important to practice good farm hygiene to control winter weeds and choose your winter rotation crop carefully. For more information on weed and crop hosts refer to 'Cotton insect pests and their weed hosts', 'Cotton insect pests and their crop hosts' and 'Managing and knowing your rotation crop' in objective 5.

For more information on the life cycle and abundance of green mirids and green vegetable bugs visit the 'Cotton Pest and Beneficial Guide' on the Australian Cotton CRC website.



It is important to consider resistance management when controlling pests such as the green mirid.

3.4.3 Using trap crops to prevent the development of resistance

Trap cropping is a technique used to concentrate a pest population into a small area of crop where they are easy to control. In cotton, trap crops can be used in spring and summer to concentrate and control populations of *Helicoverpa armigera*. This assists resistance management, as well as IPM, by reducing the size of the overall pest population which reduces the need to apply insecticides and reduces the selection pressure for the pest to develop resistance. Critical to the success of trap crops is controlling the concentrated population, preventing population increase and the flow of resistance genes. To be effective, trap crops must be considerably more attractive than the primary crop or other hosts available at the time or be the only suitable host available. If there are large areas of other attractive weeds or crop hosts, the trap crop is unlikely to be effective.

Spring trap cropping concentrates *H. armigera* moths emerging from diapause, usually between September and October. These moths will establish the first generation of larvae. The moths may originate within the cotton cropping system, i.e. they may have escaped control by pupae busting, and be carrying resistance genes from one season into the next. Alternatively, they may come from other nearby crops or weeds or be migrants from a long distance away. By concentrating and destroying this first generation it may be possible to reduce the size of future generations on cotton and other crops. Even in areas like Central Queensland, where levels of overwintering diapause is low, spring trap crops play an important role. They attract the eggs of the first and second generations of moths during a time when very few suitable hosts are available.

Summer trap cropping acts to draw *H. armigera* away from a susceptible crop like cotton, and can also produce large numbers of beneficial insects. Once the *H. armigera* are concentrated in the trap crop they can be controlled. In Central Queensland cotton growers use summer trap cropping as part of their insect resistance management strategy for Bollgard II® cotton (refer to 'Trap crops in Central Queensland' in objective 6).



Pigeon pea (a last generation trap crop) growing along side cotton.

Last generation trap cropping has also been proposed as an option to help manage resistance, though the benefits are less clear. Last generation trap crops concentrate moths emerging late in the cotton season which are the non-diapausing pupae from the last generation in autumn. These pupae are likely to be more abundant under conventional cotton and will have had intense insecticide resistance selection, so by concentrating the eggs from these moths in the trap crop, the grower can control the resulting larvae and reduce the overall number of resistant *H. armigera*. The development of non-diapausing pupae is driven by temperature. Depending on when they pupated they could emerge in the autumn or early the following spring (before the diapausing pupae). The real question then is whether last generation or autumn trap crops are really necessary or effective. In most cotton regions a high percentage of pupae formed late in the season will enter diapause (see the diapause induction Tables 11 - 14). The small number which emerge during April, May, June are unlikely to successfully generate many larvae. The most effective method to control both non-diapausing and diapausing pupae is to pupae-bust as soon as practical after harvest.

For more information on trap cropping see objective 6 - '*Using trap crops effectively*'.

3.4.4 Controlling resistant pests

The best way to reduce the population of a resistant pest is through the encouragement and use of its natural enemies (usually in combination with the absence or minimal usage of broad spectrum insecticides).

For example, a hungry ladybird can eat 50 aphids a day. Assassin bug nymphs have large appetites and can consume up to 160 small to medium sized *Helicoverpa* larvae over a 9–12 week period.

For more information on conserving beneficial insects refer to the section '*Guidelines for use of food sprays and the predator to pest ratio*' in objective 3.



A ladybird larva feasting on a large aphid colony.

3.4.5 Insecticide application failures

The presence of live pests following an insecticide application is not necessarily an insecticide failure. Some stomach poisons take 5-7 days after application before they give maximum control. Often while these insecticides are taking effect, the pest will cease feeding causing little if any economic damage to the crop.

If you suspect a spray failure:

1. Examine all the conditions that may have caused poor control.
2. Do not use rates above those recommended on the label or two insecticides of the same group. This does not improve the level of control.
3. Choose mixtures carefully on the basis of the pest spectrum.
4. If you suspect insecticide resistance, do not follow up with an application of the same insecticide group alone or in a mixture.
5. Do not expect satisfactory control of medium and large *Helicoverpa* larvae. Target *Helicoverpa* sprays against eggs and very small larvae.
6. Do not try and achieve 100% control. Aim to reduce the infestation below threshold.

For more information refer to the '*Cotton Pest Management Guide*'.

3.4.6 Resistance management for Bollgard II® crops

In Bollgard II® crops the two proteins (Cry1Ac, Cry 2Ab) that kill *Helicoverpa* and many other lepidopteran pests are present for the whole season. This prolonged exposure to the pest population of these proteins means there is very high selection for resistance.

A resistance management plan has been developed for Bollgard II® to preserve the effective life of this product. The strategy primarily makes use of the presence of two proteins as a pest would have to develop resistance to both proteins to survive (this is in theory, but in practice the two proteins are expressed at different levels and have different toxicities). The resistance strategy for Bollgard II® relies on growing refuge crops capable of producing sufficient susceptible *Helicoverpa* moths to dominate the mating with any survivors from the Bollgard II® crops as shown in Figure 10. The strategy also includes restrictions on the planting window to shorten the period of exposure to the proteins; guidelines on the management of volunteer or ratoon cotton; the destruction of pupae and control of above threshold pest levels with insecticides. In Central Queensland the destruction of pupae is ineffective as temperatures are too warm for pupae to go into diapause. Instead, trap crops are used to concentrate moths where their resulting larvae can be controlled by insecticides or pupae by cultivation.

Adherence to the resistance management plan is required under the terms of the Bollgard II® technology user agreement. For further information regarding the resistance management plan contact your Monsanto business manager or your local seed company, CSD or Deltapine.

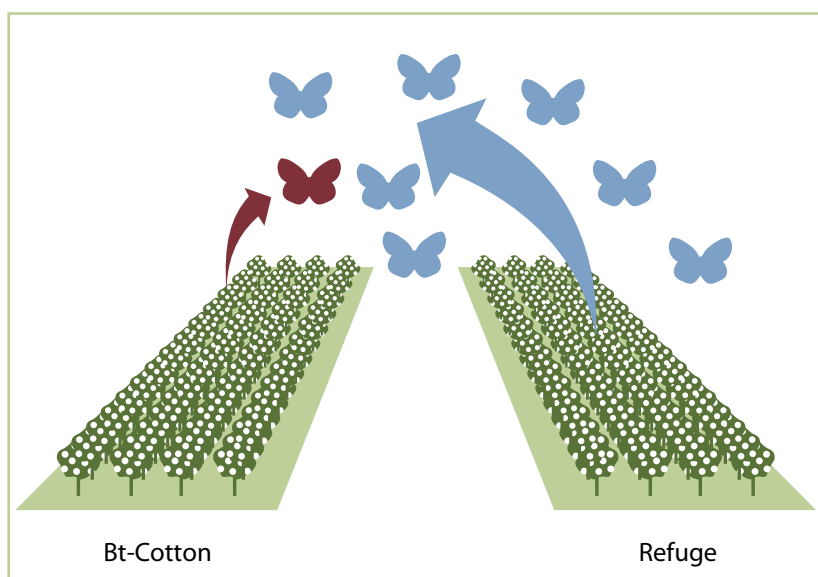


Figure 10.

Refuges produce large populations of susceptible moths (blue) to dilute any resistant moths (red) that have survived in Bollgard II® crops.

Source: Dr Gary Fitt (CSIRO)



3.5 Objective 5 - Managing crop and weed hosts

3.5.1 Cotton insect pests and their weed hosts

Many weeds are overwinter hosts for a number of pests including *Helicoverpa* spp., mites, mirids, aphids, tipworms, cutworms, armyworms and whiteflies. Poor in-field hygiene is particularly a problem with spider mites, aphids, whiteflies and mirids as these pests can move off the weeds and onto cotton seedlings in the following season. The potential for overwinter carryover of pests on weeds, and therefore for pest problems in the subsequent cotton crop, is often maximised by a mild wet winter allowing abundant growth of weeds. Ideally, management of weeds, both in fallow and cropped fields, and in field borders and headlands should be undertaken early in winter and continue through the winter as necessary. Weeds also harbour beneficials. However, the potential problems that on-farm weeds may cause, by providing overwinter hosts for pests and some diseases, generally outweighs their value as a nursery for beneficials. Growing of nursery crops for beneficials, such as lucerne, is an option available to growers who want to enhance beneficial numbers.



Bladder ketmia is the main weed host for rough bollworm.

The control of weeds also has implications for the management of cotton diseases, as some weed species are disease hosts (for more information refer to *DISEASEpak*). Similarly, good integrated weed management (IWM) principles include management of the weed seed bank. Preventing the buildup of the weed seed bank is a critical aspect of IWM and should be treated as a priority (refer to *WEEDpak* and the Farm Hygiene section of the 'Australian Cotton Best Management Practices Manual').

Most insect pests that attack cotton utilise one or more weed or native host plants. Table 16 provides a general guide to some of the weed and native plants within cotton growing regions that may also support pest populations.

Table 16. Pests and their weed hosts

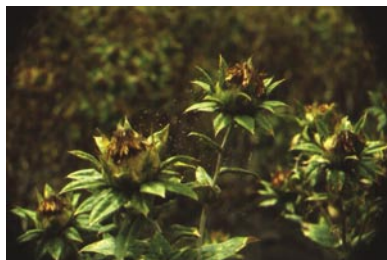
Pest name	Cotton bollworm	Native bud-worm	Spider mite	Cotton aphid	Green mirid	Apple dimpling bug	Thrips	Cotton tipworm	Rough bollworm	Silverleaf whitefly	GVB
Weed host											
African turnip											✓
Annual groundcherry				✓							
Anoda weed								✓			
Australian bindweed			✓								
Bathurst burr	✓	✓		✓							
Bellvine											✓
Blackberry nightshade			✓		✓						✓
Bladder ketmia	✓		✓						✓	✓	✓
Burr medic	✓	✓	✓				✓				
Cape honeysuckle	✓	✓									
Capeweed			✓								
Cat head	✓		✓	✓							
Cobblers peg				✓							
Common joyweed					✓						
Clover	✓	✓	✓								
Cotton regrowth	✓	✓	✓	✓	✓	✓	✓	✓	✓		
Crowfoot	✓										
Curled dock			✓								
Eucalyptus trees				✓							
Euphorbia weeds										✓	
Flax fleabane			✓	✓							
Fat-hen											✓
Gerbera	✓	✓									
Glaucia					✓						
Hairy carpet weed					✓						
Hibiscus				✓			✓				
Lucerne			✓		✓	✓	✓				
Malvastrum	✓		✓	✓	✓		✓	✓			
Marshmallow	✓		✓	✓	✓		✓	✓			
Medic	✓	✓	✓	✓							
Melon	✓	✓	✓							✓	
Mullein	✓	✓				✓					
Native rosella										✓	
Niger seed	✓	✓			✓						
Noogoora burr	✓	✓	✓		✓						✓
Paper barks				✓							
Paradoxa grass			✓								
Paddy melon			✓	✓						✓	
Paterson's curse	✓	✓		✓		✓	✓				
Pigweed				✓							
Prickly lettuce			✓								
Purple top				✓							
Saffron thistle	✓	✓									
Salt bush		✓			✓	✓					
Sida				✓							
Slender dock			✓								
Snapdragon	✓	✓									
Sow / milk thistle	✓	✓	✓				✓			✓	
Trailing verbena					✓						
Turnip weed	✓	✓	✓		✓		✓				
Variegated thistle			✓	✓	✓		✓				✓
Verbines	✓	✓									
Vetch	✓	✓									
Weeping willow				✓							
Wild sunflowers	✓	✓	✓	✓	✓		✓			✓	
Wild tobacco	✓	✓									✓
Wild turnip	✓	✓	✓	✓	✓		✓				✓
Wireweed			✓								



Sunflower crops can host a large range of pests and beneficial insects.

3.5.2 Cotton insect pests and their crop hosts

Many insect pests that attack cotton also utilise other crops as host plants. Table 17 provides a guide to the types of pests that may be supported by other crops within cotton growing regions. It is not a comprehensive list, and should be taken as a guide only.



Safflower is a good host for mites and mirids which can create problems in nearby cotton crops.

Table 17. Pests and their crop hosts

Pest name	Cotton bollworm	Native bud-worm	Spider mites	Cotton aphid	Green mirid	Apple dimpling bug	Thrips	Cotton tipworm	Rough bollworm	Silverleaf whitefly	GVB
Crop host											
Adzuki bean	✓	✓	✓		✓						✓
Barley	✓						✓				
Bean	✓	✓	✓		✓		✓				
Cabbage											✓
Canola	✓	✓	✓	✓			✓				
Capsicum				✓							✓
Chickpea	✓	✓									
Corn			✓								✓
Cotton	✓	✓	✓	✓	✓	✓	✓	✓	✓	✓	
Cowpea	✓	✓			✓						✓
Cucurbits			✓	✓						✓	✓
Faba bean	✓	✓	✓	✓	✓		✓				
Field pea	✓	✓	✓								
Hop			✓								✓
Lablab	✓	✓	✓	✓	✓	✓					✓
Lettuce											✓
Linseed	✓	✓									
Lucerne	✓	✓	✓		✓	✓	✓				✓
Lupin	✓	✓			✓						✓
Melon	✓	✓	✓	✓						✓	
Mungbean	✓	✓	✓		✓					✓	✓
Native bean											✓
Navy bean										✓	✓
Peanut	✓	✓	✓							✓	
Pecan nut											✓
Pigeon pea	✓	✓		✓	✓	✓					
Safflower	✓	✓	✓		✓	✓					
Sesame	✓	✓									✓
Sorghum	✓		✓	✓		✓					
Soybean	✓	✓	✓							✓	✓
Sunflower	✓	✓	✓	✓	✓	✓				✓	✓
Tomato	✓	✓	✓							✓	✓
Wheat	✓			✓			✓				

3.5.3 Managing cotton stubble re-growth

Cotton re-growth after harvest can provide a food source for *Helicoverpa* spp., spider mites, green mirids, apple dimpling bugs and aphids. Cotton re-growth can carry these pests between seasons or infest other crops planted into the same field, including cotton. Re-growth should be controlled by slashing or by use of a defoliant.

Managing cotton re-growth is also important in terms of disease carry over. It is a risk for carry over of the disease Cotton Bunchy Top (CBT). If cotton plants become infested with aphids late in the season there is a chance that they will be infected by CBT, though symptoms will not show before defoliation. If allowed to survive through winter, these plants will re-grow with CBT symptoms in the following spring. Cotton aphids feeding on these plants could then pick up CBT and spread it to adjacent cotton crops. For more information refer to *DISEASEpak*.

Further, control of cotton re-growth is an important component in resistant management for Bollgard II® crops, and for Round-up Ready® varieties (for further information see the ‘*Cotton Pest Management Guide*’, *WEEDpak* and the Farm Hygiene section in the ‘*Australian Cotton Best Management Practices*’ manual).



Cotton regrowth in a Faba bean crop. Cotton regrowth is a potential over-winter host for many pests and for the disease Cotton Bunchy Top and should be controlled.

3.5.4 Managing and knowing your rotation crop

An important part of any cropping system is the rotational phase. Rotation crops may be planted for a number of reasons, including favourable price, improving soil structure and nitrogen fixation (legumes). The selection of a rotation crop also has implications for pest management. Some may also affect carry over of disease as suggested in *DISEASEpak*. The choice of rotation crop should take these issues into account.

Options for managing pests in rotation crops should also be considered. For instance, use of broad spectrum insecticides to manage *Helicoverpa* or aphids in rotation crops may have detrimental effects on beneficial insect populations and hence reduce the number later moving into the cotton or nursery crops. Similarly, control of mirids in safflower often results in outbreaks of mites, which then move into adjacent cotton crops requiring expensive control.

A suitable rotation crop can provide numerous benefits which may or may not be apparent. Cereal crops have the advantage of breaking disease cycles and some legume crops can improve soil structure and nutrition. However the growth of various rotation crops vary with latitude, so local experience and some interpretation of research trials from different regions is required. For example, ‘Highworth’ lablab will not flower in the Namoi valley, but will flower in the Darling Downs thus it could have the potential to create a weed problem. The decision to choose a rotation crop that best fits your system must consider many factors, interactions, advantages and disadvantages as shown in Table 18.

In terms of IPM, many rotation crops act as a host for a range of pests, such as faba beans (mites, aphids), safflower (mites, mirids), chickpeas (*Helicoverpa* spp.) or cereals (*H. armigera* and thrips). This has been covered in the section on ‘*Cotton insect pests and their crop hosts*’ in this objective. Growing a range of crops can be seen as essential to providing a habitat for a variety of beneficial insects. Cotton in monoculture over a wide area provides little opportunity for beneficials to thrive and persist.



Growing a range of crops can be seen as essential to providing a habitat for a variety of beneficial insects.

Table 18. The advantages and disadvantages of winter rotation crops

	Insects	Weeds	Diseases	Soil
Chickpeas	Excellent trap crop for <i>Helicoverpa</i> . Should check crop regularly and must pupae bust after harvest. Not a good nursery crop for <i>Trichogramma</i> and most beneficials.	There are limited broad-leaf herbicide options and be careful not to restrict options for the following summer crop.	May decrease the risk of bacterial blight and alternaria leaf spot but may increase the chance of verticillium, black root rot and fusarium (depending on season and crop residue).	Is a deep rooted crop (in non sodic soils) and can improve aggregate stability. Can also fix up to 150 kg / ha of N. Can improve soil penetrability. Excellent N fixer (up to 300 kg / ha) and can accumulate K and reduce premature senescence. Can grow in sodic soils. Encourages VAM*.
Faba beans	Nursery crop for <i>Helicoverpa</i> and beneficial insects. Can aggravate mites and aphids.	There are limited broadleaf herbicide options, volunteer faba beans may be a problem and can increase sowthistle populations in the following cotton crop.	Disease management in the faba bean crop is essential. May decrease the risk of bacterial blight, but could increase the risk of all other major cotton diseases.	Is a deep rooted crop (in non sodic soils) and can improve aggregate stability. Doesn't grow well in sodic soils. Very effective N fixer (up to 270 kg / ha)
Vetch	Nursery crop for beneficial insects.	Limited herbicide options. Weeds and volunteer vetch is not a problem if mulched adequately.	Can decrease the risk of black root rot, but if crop residues are incorporated late it may increase the risk of seedling disease. Increases severity of fusarium wilt.	Can improve soil penetrability. Excellent N fixer (up to 300 kg / ha) and can accumulate K and reduce premature senescence. Can grow in sodic soils. Encourages VAM.
Lucerne	Nursery crop for beneficial insects, mirids, <i>Helicoverpa</i> , mites and aphids.	Helps in the management of nutgrass (by drying soil profile). Weeds relatively easy to control although difficult to destroy lucerne plants in cotton.	May increase the risk of phytophthora boll rot, black root rot, fusarium and sclerotinia. If crop residues are incorporated late it may increase the risk of seedling disease.	Encourages VAM.
Canola	May aggravate mites, mirids, aphids, rutherghlen bugs and <i>Helicoverpa</i> .	Herbicide tolerant varieties are available. There are limited broadleaf herbicide options for conventional varieties.	May decrease the risk of bacterial blight, verticillium wilt and black root rot.	This crop is has no mycorrhizal activity and therefore cotton is rarely grown after canola. The tap root can improve soil structure.
Safflower	May aggravate mites, mirids, thrips, rutherghlen bugs and <i>Helicoverpa</i> .	There are limited broadleaf herbicide options and late broad leaf weeds can be a problem, particularly sowthistle.	May increase the risk of verticillium wilt, but can decrease the risk of bacterial blight and seedling diseases.	The roots dry soil to 2 m. Pre-irrigation following a safflower crop penetrates to about 1 m, leaving a dry band from 1-2 m, preventing deep drainage in subsequent crops. The tap root can break through hard layers.
Cereals (wheat, barley) etc	May be an increased risk of thrips and wireworms.	Generally a decrease in weed pressure.	Excellent break for all diseases, except for fusarium.	Breaks down slowly for slow release of carbon. Reduces run-off and erosion, improves soil structure.

* VAM, Vesicular Arbuscular Mycorrhiza. In most soils, cotton is dependent on VAM for successful growth. VAM fungi colonise roots and surrounding soil and act as an extension of the root system by supplying extra phosphorus and zinc. In return the plant 'feeds' the VAM fungi with sugars produced in the leaves. Cotton growth is reduced and maturity may be delayed when there are insufficient VAM fungi in the roots. Seedlings are stunted with small leaves and short internodes and phosphorus and zinc deficiency symptoms may be obvious.

The Cotton Research and Development Corporation together with the Australian Cotton CRC Farming Systems Extension Team have produced a poster titled '*Rotation crops and cotton*' which details the advantages and disadvantages of all possible rotation crops in summer and winter (in relation to the following cotton crop). Pick up your poster from your local industry development officer or from the TRC.



3.6 Objective 6 - Using trap crops effectively

3.6.1 Introduction

Trap cropping is an IPM tactic that can be utilised on a farm level or an area wide basis. Trap cropping is essentially a method of concentrating a pest population into a manageable area by providing the pest with an area of a host crop or an area of a preferred host crop. When strategically planned and managed, trap crops can be utilised at different times throughout the year to help manage a range of pests. In particular, utilising trap crops in the spring and summer has become an integral component of IPM in area wide groups.

Spring trap crops

Spring trap crops are designed to attract *Helicoverpa armigera* as they emerge from overwintering pupae in spring. A trap crop, strategically timed to flower in the spring, can help to reduce the early season buildup of *H. armigera* in a district.

Spring trap cropping, in conjunction with good *Helicoverpa* control in crops and pupae busting in autumn, is designed to reduce the size of the local *Helicoverpa* population. Over time, this strategy is anticipated to reduce *Helicoverpa* pressure on susceptible crops in the participating region.

This approach is used in different ways:

1. In southern areas, where there is a high incidence of overwintering *H. armigera*, an area of flowering trap crops acts to concentrate locally emerging *H. armigera* moths into a crop where they can be destroyed. The main period of emergence is September to October.
2. In Central Queensland there is minimal overwintering of *Helicoverpa* because temperatures are generally too warm to trigger diapause. Here spring trap crops are used to concentrate local *H. armigera* populations into areas where they can also be destroyed, at a time when there are few other hosts for the *Helicoverpa*.

Summer trap cropping

Summer trap cropping has quite a different aim from that of spring trap cropping. A summer trap crop aims to draw *Helicoverpa* away from a main crop such as cotton or mungbean and concentrate them in another crop such as sorghum, pigeon pea or lab lab. Once concentrated into the trap crop, the *Helicoverpa* larvae can be controlled. In Central Queensland cotton growers are using summer trap crops of pigeon pea as part of their insect resistance management strategy for Bollgard II® cotton.

Some summer trap crops may produce large numbers of beneficial insects that can then move into nearby crops, for example, the parasitic wasp *Trichogramma* in sorghum and maize.



Helicoverpa large larva, controlled using a virus in a pigeon pea trap crop.

3.6.2 First generation or 'Spring' trap crops in southern areas

An ideal first generation trap crop is very attractive to *H. armigera*, is a good nursery for beneficials, does not host secondary pests or diseases, does not become a weed problem and is easy to establish and manage.

Many winter crops have been trialed to measure their potential as a spring trap crop. Chickpea has consistently proven superior to all other crops in its ability to generate large numbers of *H. armigera*, however it is not a good nursery for beneficial insects. Chickpea has also proven to be agronomically robust, suitable for both dryland and irrigated situations.

Early season trap crops ideally direct eggs from *H. armigera* moths emerging from their overwintering diapause. These moths are the carriers of resistance from one season to the next. The trap crops must therefore be attractive from early October onwards.

Growers must ensure trap crops do not become future nurseries of *Helicoverpa*, and so effectively controlling populations in the trap crop by timely destruction of the crop itself is required. Because the trap crop will not be harvested for yield a fast knock-down insecticide is not required, therefore bio-pesticides like Bt and virus formulations may be well suited. In some seasons spring trap crops may be overwhelmed by *H. punctigera* and migrant *H. armigera* prior to the local emergence of moths from diapause.

Be mindful when selecting the type of trap crop to plant, as some effective trap crops such as field pea can also be a host for mirids.

More information on spring trap crops can be found in the publication '*Spring trap crop management guidelines*', which is available from the TRC, or from '*Trap-cropping: A fad or a useful heliothis management tool?*' available on the DPI&F Queensland website.

3.6.2.1 Managing chickpea as a spring trap crop

Chickpea has proven to be agronomically robust and suitable for both dryland and irrigated situations. However the search for an alternative to chickpea has been driven by its problems, i.e. *Ascochyta* (a disease) control requirements and the lack of beneficial insect production in chickpea. Many crops have been assessed over the past few years including; field pea, canary, linseed, canola, mustard, niger, early sunflower, lentil and vetch. In all trials, chickpea recorded the highest *Helicoverpa* numbers, whilst many of the other crops were hosts for secondary pests such as mirids and green vegetable bugs.

When to sow

Strong healthy flowering crops are more attractive to *Helicoverpa* than non-flowering crops. For example, the recommended planting time for chickpea trap crops in Central and Southern Queensland is late July to early August. This will ensure that the trap crop is most attractive to *Helicoverpa* when the moths emerge from their winter diapause in late October - November. Check Table 19 to find the ideal planting time.

Table 19. lists the date at which 50% of plants will flower for a range of planting dates at each of the locations given for the Amethyst chickpea cultivar. It is based on long term average temperatures, and so should be considered as a guide only. Some plants will commence flowering earlier than the dates given, and generally the day on which 5% of plants are in flower falls about 5 days earlier than the 50% date. The dates at which 95% of plants will be in flower are generally 5 to 10 days after the 50% date. Amethyst chickpea will continue flowering whilst soil moisture is available.



In all trials, chickpea recorded the highest *Helicoverpa* numbers.

Table 19. The date at which 50% of Amethyst chickpea plants will have commenced flowering for a range of planting dates and regions.

Planting Date	Gunnedah	Narrabri	Walgett	Moree	Goondi-windi	St. George	Dalby	Emerald
1st June	31-Aug	27-Aug	26-Aug	27-Aug	24-Aug	5-Aug	9-Aug	30-Jul
15th June	10-Sep	7-Sep	6-Sep	7-Sep	4-Sep	22-Aug	25-Aug	16-Aug
1st July	21-Sep	18-Sep	16-Sep	17-Sep	14-Sep	2-Sep	5-Sep	31-Aug
15th July	29-Sep	26-Sep	24-Sep	25-Sep	22-Sep	15-Sep	18-Sep	8-Sep
1st Aug	8-Oct	5-Oct	3-Oct	3-Oct	2-Oct	24-Sep	28-Sep	21-Sep
15th Aug	16-Oct	13-Oct	11-Oct	12-Oct	10-Oct	2-Oct	4-Oct	1-Oct
1st Sept	25-Oct	23-Oct	21-Oct	22-Oct	20-Oct	14-Oct	15-Oct	15-Oct
15th Sept	3-Nov	2-Nov	30-Oct	31-Oct	30-Oct	24-Oct	26-Oct	25-Oct
1st Oct	13-Nov	11-Nov	10-Nov	11-Nov	10-Nov	6-Nov	7-Nov	5-Nov
15th Oct	24-Nov	22-Nov	20-Nov	22-Nov	21-Nov	16-Nov	17-Nov	15-Nov

Size of trap crop

Research results have shown that blocks are more effective than strips or patches. It is recommended to plant a minimum of 2 hectares or 1% of the total cultivated area.

Where to plant the trap crop

To lessen the risk of the major chickpea disease *Ascochyta*, do not plant the chickpeas in or near old chickpea stubble. If you are planting commercial chickpeas, then plant your trap crop near those crops to simplify rotations and chemical applications. You can use some of your commercial chickpea crop as a trap crop, by slashing to prolong flowering until October.

Planting

The recommended plant stand is 20 / m². This rate is lower than crops planted commercially as the trap crop will not be harvested and lower populations will produce larger plants with a prolonged flowering period.

To promote a healthy plant, an inoculant and starter fertiliser should be used along with a seed treatment for *Ascochyta*. Treatments for various diseases such as *Botrytis* grey mould, *Phytophthora* and *Pythium* are also recommended, along with selecting a variety with disease tolerance.

Fungicides

The management of the chickpea disease *Ascochyta*, relies on program applications of fungicides. Even if the crop appears free of the disease, early application of a fungicide is critical in restricting its development.

Crop scouting

Once established, chickpea trap crops should be regularly checked on a weekly basis for insects and diseases. Visual checks are the best method to look for eggs, small larvae and the presence of any disease. Each visual check can be carried out by scouting 5 chickpea plants within one meter. This should be repeated at least 6 times at random sites throughout the crop.

Using insecticides on the trap crop

Spraying maybe required to protect the trap crop from large infestations of *H. punctigera* if they occur, to prolong its usefulness and to make sure that it does not become a nursery for *H. Punctigera*. If spraying is necessary, consider using biological products like Gemstar® or Dipel SC®. This will ensure that there is no pre-selection for resistance, and minimal disturbance to beneficial insects in the area.

Destroying trap crops

To avoid creating a nursery for *H. armigera*, the trap crop must be destroyed prior to the pupation of the first large *H. armigera* larvae. To destroy the trap crop, use at least one full disturbance cultivation. To do this, slash the crop before ploughing it out using a disk or chisel plough so that the stubble is completely incorporated.



Checking a chickpea crop.

For more information refer to the ‘*Spring Trap Crop Guidelines*’ available from the TRC.

3.6.3 Trap crops in Central Queensland

In Central Queensland there is minimal overwintering of *Helicoverpa* because temperatures are generally too warm to trigger diapause. Here spring trap crops are used to concentrate local *H. armigera* populations into areas where they can also be destroyed, at a time when there is a natural gap in the availability of host plants for *H. armigera*.

Bollgard II® requirement

A requirement for Central Queensland in the guidelines for pest management in Bollgard® II cotton, suggests that 1% of the Bollgard® II cropping area should be planted to a chickpea trap crop. This crop must be monitored regularly for *Helicoverpa*, and if substantial numbers of large larvae are found during the last half of September, the crop should be ploughed down as soon as possible. If not, all chickpea trap crops should be effectively cultivated by the 30th September.

Summer trap crops

There is considerable interest in summer trap cropping, which has quite a different aim from that of spring trap cropping. A summer trap crop aims to draw *Helicoverpa* away from a susceptible crop such as cotton or mungbean and concentrate them in another crop such as sorghum, pigeon pea or lab lab. Once concentrated in the trap, the *Helicoverpa* larvae can be rigorously controlled or the crop destroyed. In addition, some summer trap crops may produce large numbers of beneficial insects that can then move into nearby crops, for example, the parasitic wasp *Trichogramma* in sorghum and maize. In Central Queensland cotton growers are using summer trap crops of pigeon pea as part of their insect resistance management strategy for Bollgard II® cotton.

The summer trap cropping including in-season and end of season concepts, are still in a research phase and any benefits are not yet quantified. At this stage, summer trap cropping for *Helicoverpa* (other than in Central Queensland) is not being promoted as a tool for *Helicoverpa* management.

The diversity of the cropping system on the Darling Downs, provide a ‘natural’ system of trap crops. An alternative approach to specifically planting summer trap crops, is to manage the current range of crops (particularly winter cereals, sorghum and maize) to act as a trap or a beneficial nursery.

For more information on trap cropping visit the DPI&F, Queensland website.

3.6.4 Last generation trap crops

The aim of a ‘last generation’ trap crop is to attract moths emerging from non-diapausing pupae under cotton. These pupae will have had intense insecticide resistance selection on the cotton crop. Concentrating the eggs from these moths in the trap crop allows the resulting larvae to be controlled using biological insecticides such as a virus or by cultivation to kill the resulting pupae. This reduces the number of moths contributing to the next generation which contributes to resistant management. However, in practice the benefits of the last generation trap crops are less clear (refer to ‘*Using trap crops to prevent the development of resistance*’ in objective 4).

The trap crop would be planted in December to January in eastern Australia, to ensure that it was highly attractive to *H. armigera* late in the cotton season. The trap crop must be sufficiently attractive to change the local distribution of *Helicoverpa* moths, drawing them into the trap crop area and effectively concentrating egg laying in the trap crop. The attractiveness of the cotton crop relative to the trap crop may significantly influence the potential effectiveness of this strategy. The eggs and larvae in



Pigeon Pea can be used as an attractive last generation trap crop.

the trap crop can be destroyed using biological sprays, and the pupae controlled using cultivation. Care is needed to ensure that the trap crop doesn't become a nursery for pests. The area of trap crop planted is also important, with most area wide management groups using 1% of the total farm area as a starting point. This area can be reused to plant the spring trap crop (discussed earlier). As yet however there are no data to justify that 1% is sufficient.



3.7 Objective 7 - Supporting IPM through communication and training

3.7.1 Communicate with neighbours

Communication with neighbouring primary producers is essential to develop a successful IPM program. During the season when there is a possibility that crop spraying will occur, it will be important to discuss your intentions and options with your neighbours as a safety precaution and to help manage the crop as part of a large area not just as a block. It is just as important to communicate with non-cotton growing neighbours as well, and if possible encourage your neighbours to reciprocate a level of communication.



Attending field days can be a great way to learn about IPM, hear about the latest research findings and catch up with neighbours.



Growers discussing management plans for the season.

3.7.2 Pesticide application management plan

Growers should discuss their approach to crop management with their agronomist or consultant. Mutual agreement on the IPM approach between the grower and consultant, and awareness by the grower of the reasons underlying this approach and of the need to view pest management differently is essential for success. The grower and their consultant need to work as a team. A consultant cannot be expected to manage pests according to IPM principles if the grower expects unrealistic yields and levels of pest control. The grower and consultant should have a pre-season meeting to discuss issues such as yield and crop maturity expectations, thresholds for pests, crop and insect monitoring and spray management plans.

The grower should also discuss the spray management plan with the applicator as well as neighbours. An important issue to discuss with the spray applicator is the hygiene of spray equipment, including ground rigs and airplanes. Residues of broad spectrum insecticides in the tanks or sumps of spray equipment can contaminate selective products causing undesirable and unintended detrimental effects on beneficial insects. Growers should ensure that equipment is thoroughly cleaned out. More information on managing the risks associated with pesticide use can be found in the 'Australian Cotton Pest Management Practices' manual.

Issues such as when to spray insecticides, application requirements (swath width, water volume etc) and fields in sensitive locations should also be discussed.

Growers should consider discussing with their neighbour(s) the possibility of amalgamating as part of an IPM group or, alternatively, the growers should discuss the problems associated with the use of broad spectrum sprays such as organophosphates and pyrethroids early in the season and their impact on beneficial insects. For some insecticides, specific 'buffer' requirements apply and it is critical that growers and consultants discuss this issue and consult with neighbours.

3.7.3 Area Wide Management (AWM)

AWM primarily attempts to reduce pest pressure by co-ordinating the efforts of growers in an area, for example, to use trap crops planted in spring to capture *Helicoverpa armigera* moths into an area where the larvae developing from their eggs can be readily controlled. This prevents breeding and population increase, thereby reducing pest pressure later in the season. This is a form of pest population management.

AWM is also used to describe situations where growers share information and support each other to achieve common goals. For instance, a goal may be to delay the use of disruptive insecticides as late as possible in the season, so that the survival rate of beneficial insects is higher and can contribute to pest control.

AWM groups or IPM groups, as they are sometimes known, use an approach which acknowledges that pest and beneficial insects are mobile, and that the management regimes to control pests imposed on a given field are likely to alter the abundance of beneficial insects and levels of insecticide resistance in the surrounding locality, e.g. the effect of spray drift on beneficials in a neighbour's crop. By communicating and co-ordinating strategies, growers within an AWM group have better opportunities to implement IPM.

AWM for population management

AWM for population management of *H. armigera* has been evaluated on the Darling Downs. The strategy encouraged all farmers to work cooperatively and take a regional, rather than a paddock-by-paddock approach to pest management. With this approach, it was thought that the size of the local *H. armigera* population could be reduced, giving non-chemical tools such as virus and beneficials a greater chance of being effective.

This strategy was based on three main goals: (1) to reduce the survival of overwintering, insecticide resistant *H. armigera* pupae, (2) to reduce the early season buildup of *Helicoverpa* on a regional / district scale, and (3) to reduce the mid-season population pressure on *Helicoverpa* susceptible crops. The main tactics used to achieve these goals were use of spring trap crops, conservation of beneficial insects and cultivation of diapausing pupae. A critical component was to bring together farmers with a range of different enterprises, including cotton, wheat and other dryland crops. As *H. armigera* is a pest common to most of these crops it was vital to have all types of growers involved if AWM was to succeed.

The spring trap crops and pupae busting essentially targets 'bottlenecks' in the *Helicoverpa* population cycle. For instance spring trap crops aim to provide an attractive crop for *H. armigera* emerging from diapause at a time when there are often relatively few hosts (refer to objective 6 'Using trap crops effectively'). Similarly pupae busting attacks the diapausing pupae in the soil where they are vulnerable and where there is a fairly wide window of opportunity in which to act (refer to the *Helicoverpa* section in objective 4).

Success at manipulating *H. armigera* abundance is difficult to assess due to the confounding effects of seasons (e.g. drought), but certainly in the years following the implementation of AWM there has been reduced *H. armigera* pressure.

AWM or IPM Groups

These groups focus less on pest population management and more on communication and co-ordination to achieve agreed goals, which are usually linked to increasing the adoption of IPM. These may include conserving beneficials, delaying the use of disruptive insecticides, reducing the risk of drift between farms and the planting trap crops. A key element of groups that have worked well has been regular meetings before and during the season to share information, agree on goals, discuss strategies and build rapport.



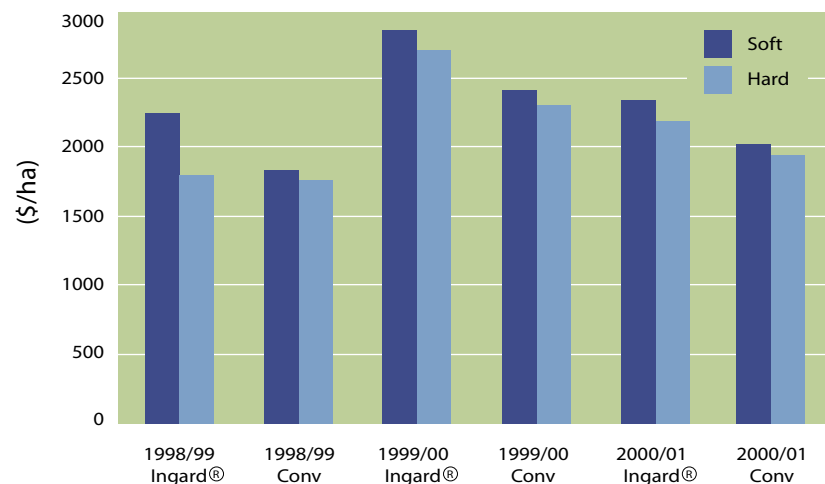
Area Wide Management (AWM) recognises that actions on one farm can affect another, therefore there are advantages in communication and coordination.



'Hands-on' group activities are an effective way to learn.

Figure 11.

Gross margins for the Boggabilla Area Wide Management Group over three seasons for a field managed with more selective insecticides (Soft) or more disruptive insecticides (Hard). Fields managed with a softer approach were consistently more profitable.



Getting an AWM group going (formation and operation)

When forming an AWM group there are some things that should be carefully thought through.

- Decide which area and who will be involved, i.e. growers, consultants, agronomists, contractors
- Set goals
- Identify specific activities for the group, i.e. avoid meeting for meetings sake
- Keep notes to use for revising and resetting goals and to keep non-attendees informed.

Activities for groups with minimal support can include:

- Using pheromone traps to generate information about *Helicoverpa* activity (species and time) in the local area. This information may help form a basis to determine the appropriate time for planting and destruction of trap crops.
- Seeking guest speakers
- Gather and discuss pest and beneficial pressure and management options.

Contributions that your cotton industry development officer (IDO) can make with out being the group co-ordinator:

- Introduce the group to new IPM options and techniques.
- Challenge the attitudes and perceptions of the group.
- Use their network in the cotton industry to gather information for the group.

Be aware of group development processes. Most groups go through the stages of forming, storming, norming and performing. *Forming* is the beginning stages when people are sorting out their place in the group and how things will run. *Storming* is the time when group members may experience conflict as individuals resist the influence of the group and oppose group direction and consensus. *Norming* occurs when group cohesiveness and commitment is achieved. The participants discover ways to work together to achieve goals and objectives. Finally, the *performing* stage is when a group develops proficiency in reaching the desired goals and is also more flexible about ways of working together. It is useful to realise that all groups go through these stages.

Initially some people will be drivers and innovators and others will take more time to work effectively together. It is important to ensure that each individual is given the opportunity to share their ideas and concerns. Every group is unique, and every group will develop a different set of goals and priorities.

Maintaining momentum

Maintaining momentum, interest and learning are major challenges for AWM groups. After groups have been operating for two to three seasons they can lose momentum if they have not continued to provide a stimulating learning environment. To help prevent this, regularly rotate the chairperson and secretary positions to keep all members involved and to ensure everyone has a sense of ownership. Groups should also embark on a process of continual improvement by revising and resetting goals.

Another way to maintain group momentum is to benchmark IPM strategies and outcomes. This will allow group members to see which strategies worked best for them and how they compared with the rest of the group. This is an effective way to identify the key factors contributing to success and areas for improvement.

When not everyone wants to be involved

AWM groups are voluntary and not all growers may want to be involved. However, as groups develop, the “non-participants” may see the benefits of the group in action and be encouraged to join.

If a neighbour does not want to participate initially, ensure that they are given the opportunity to receive meeting notices and information about meeting outcomes and group goals.

Conclusion

AWM groups are a tremendous way to support IPM by providing peer support, defining goals, communicating and enlisting expert input when needed. The progress made by many AWM groups in reducing insecticide use and increasing profitability attests to their value.

3.7.4 Meetings and training

Meetings are held each winter in all major regions to review resistance levels, IPM principles, computerised decision support, Best Management Practice procedures, production issues and to evaluate the previous cotton crop. Growers and consultants are urged to participate in these meetings. This improves information exchange between industry, research and extension and facilitates improvements in IPM strategies.

Growers and consultants should also consider formal training in IPM principles and practice. Two options developed by the Australian Cotton CRC are the cotton production course offered by the University of New England along with several other universities, and / or the IPM short course for growers.

For more information on the Australian Cotton CRC training courses, visit the Australian Cotton CRC website or contact cotton production course co-ordinator, at the University of New England, or contact the IPM short course training co-ordinator, at the DPI&F, Queensland.



Demonstrating the beat sheet technique.

Glossary

Aphid colony	4 or more aphids within 2 cm on a leaf or terminal.
Area Wide Management (AWM)	Growers working together in a region to manage pest populations. AWM is a cotton industry vehicle driving adoption of on-farm IPM.
At-planting insecticide	Insecticides applied in the seed furrow with the seed during planting. The insecticide may be applied as a granule or as a spray into the seed furrow.
BDI	Beneficial Disruption Index – the sum of scores for the entire cotton season of the impact of each insecticide on beneficial insect populations. The BDI helps benchmark the ‘softness’ or ‘hardness’ of an individual fields’ insecticide spray regime.
Beat sheet	A sheet of yellow canvas 1.5 m x 2 m in size, placed in the furrow and extended up and over the adjacent row of cotton. A metre stick is used to beat the plants against the beat sheet. Insects are dislodged from the plants onto the canvas and are quickly counted.
Beneficial insects	Predators and parasitoids of pests.
Biological insecticides	Insecticides based on living entomopathogenic (infecting insects) organisms, usually bacteria, fungi or viruses, or containing entomopathogenic products from such organisms i.e. Gemstar, Vivus and Dipel (BT).
Boll	Cotton fruit after the flower has opened and fertilisation has occurred (after the flower has turned pink). Bolls typically have four or five segments, known as locks, each containing about 6 - 10 seeds. The lint, or cotton fibre, is produced by elongated cells that grow from the surface of the seed coat, hence the ‘seed cotton’ in the boll is a mixture of seed and lint.
Bollgard II® cotton	Genetically modified cotton variety containing the insecticidal proteins Cry1Ac and Cry2Ab which provides control of <i>Helicoverpa</i> spp., rough bollworm, cotton tipworm and cotton looper under field conditions.
Broad spectrum insecticide	Insecticides that kill a wide range of insects, including both pest and beneficial species. Use of broad spectrum insecticides usually reduces numbers of beneficials (predators and parasites) leading to pest resurgence (see below) and outbreaks of secondary pests.
Buffer zone	A boundary of land or crop set up within or outside the cotton farm to collect spray droplets that may otherwise drift onto sensitive areas, such as rivers or pasture.
Calendar sprays	Insecticides sprayed on a calendar basis, e.g. every Friday, regardless of pest density or the actual need for pest control.
Cold shock	Is when the daily minimum temperatures fall below 11°C. When this occurs, cotton growth and development the following day can be reduced regardless of the maximum temperature reached. Cold shocks have greatest impact on early plant development and will delay the timing of emergence, squaring and flowering and increase the susceptibility of plants to diseases.
Consecutive checks	Refers to successive insect checks taken from the same field or management unit.
Conventional cotton	Strictly a cotton variety that does not contain transgenes (genes from other species), but used in this guide to indicate varieties that do not include genes to produce insecticidal proteins (i.e. Bollgard II®) but which may include herbicide resistance genes (i.e. Round-up Ready®).
Cotton bunchy top (CBT)	A relatively new disease spread by the cotton aphid (<i>Aphis gossypii</i> , Glover). Symptoms of CBT include reduced plant height, leaf surface area, petiole length and internode length. Pale angular mottling of the leaf margins is the most reliable diagnostic feature.
CottonLOGIC	A suite of software packages developed by the Australian Cotton CRC for the Australian cotton industry which includes EntomoLOGIC, HydroLOGIC and NutriLOGIC.
Cotyledons	Paired first leaves that emerge from the soil when the seed germinates.
Crazy cotton	Multi-branched cotton caused by excessive and repeated tipping out.
Crop compensation	The capacity for a cotton plant to ‘catch-up’ after insect damage without affecting yield or maturity.
Crop maturity	This usually occurs when 60-65% of bolls are mature and open. Cotton bolls are mature when the fibre is well developed, the seeds are firm and the seed coats are turning brown in colour.
Cut-out	As the cotton plant continues to develop bolls, the demand for carbohydrates that are produced in the leaves increases. Eventually the demand by the bolls exceeds supply, resulting in the production of new fruiting nodes ceasing and the shedding of excess bolls, less than 14 days old. This point is known as “cut-out.” An approximation of the timing of cutout is when a crop has reached on average 4 nodes above white flower (NAWF).

Damage threshold	The level of damage from which the crop will not recover completely and which will cause some economic loss of yield or delay in maturity. Damage thresholds are usually applied in conjunction with pest thresholds to account for both pest numbers and plant growth. For instance a plant which has a very high fruit retention (see below) may be able to tolerate a higher pest threshold (see below) than a crop with poor fruit retention.
Day Degrees (DD)	A unit combining temperature and time, useful for monitoring and comparing crop development. To calculate your DD visit the Australian Cotton CRC website.
Deep drainage	Water from rainfall or irrigation that has drained below the root zone of the crop. A certain amount of deep drainage helps flush salts from the soil, but excess deep drainage means water and nutrients are being wasted.
Defoliation	The removal of leaves from the cotton plant in preparation for harvest. This is done by artificially enhancing the natural process of senescence and abscission with the use of specific chemicals.
Denitrification	A biological process encouraged by high soil temperatures. Denitrification occurs when there is waterlogging, such as during and after flood irrigation and/or heavy rainfall sufficient. The process converts plant available N (nitrate) back to nitrogen gases which are lost from the system.
Diapause	A period of physiologically controlled dormancy in insects. For <i>Helicoverpa armigera</i> , diapause occurs as the pupal stage in the soil.
D-Vac	A small portable suction sampler or blower / vacuum machine used to suck insects from the cotton plants into a fine mesh bag. D-vac samples are collected by passing the tube of the vacuum sampler across the plants in 20 m of row. When plants are small this may be a single pass, but when plants are bigger a zig zag pattern from the bottom to the top of the crop with each step of the operator may be required to sample the canopy more effectively. Samples from the d-vac bag are transferred into a plastic bag and counted.
Earliness	Minimising the number of days between sowing and crop maturity. Within a cotton variety earliness usually involves some sacrifice of yield.
Early season diagnostic (ESD) tool	A web-based tool to graph and display day degrees and node counts against a theoretical optimum crop development rate to determine where the crop development is at compared with where it should be.
Efficacy	The effectiveness of a product against pests or beneficial insects (predators or parasites).
Egg parasitoids	They are parasitoids that specifically attack insect eggs. E.g. <i>Trichogramma pretiosum</i> attacks the egg stage of <i>Helicoverpa</i> . The wasp lays its eggs in the egg, and the wasp larvae which hatch consume the contents of the host egg. Instead of a small <i>Helicoverpa</i> larva hatching, up to four wasps may emerge from each host egg. Thus the host is killed before causing damage.
EntomoLOGIC	Pest management software available from CSIRO and the Australian Cotton CRC.
Flat fan nozzle	A spray nozzle with an outlet that produces spray droplet distribution that spreads out of the nozzle in one direction but which is thin in the other direction, much like the shape of a Chinese or Japanese hand fan.
Flush	A high volume irrigation carried out in minimal time.
Food sprays	They are natural food products sprayed onto cotton crops to attract and hold beneficial insects, particularly predators, in cotton crops so they can help control pests. Two types of food sprays are available for pest management. They are the yeast based food sprays which attract beneficial insects and the sugar based ones which retain predators which are already in the crop.
Fruit load	Refers to the number of fruit (squares or bolls) on a cotton plant.
Fruit retention	Refers to the percentage of fruit (squares or bolls) that the cotton plant or crop has maintained compared with number it produced.
Fruiting branch	Grows laterally from the main stem in a series of segments. Each segment finishes at a node at which there is a square and a leaf. At the base of the square next segment originates, and so on.
Fruiting factor	Is a measure of the number of fruit per fruiting branch. A method to check if the total fruit number produced by the crop is on track. Fruiting factors which are too high or too low can indicate problems with agronomy or pest management which may need to be acted on. To calculate the fruiting factor divide the fruit count made in 1 metre of cotton row by the number of fruiting branches in that area.
Habitat diversity	A mixture of crops, trees and natural vegetation on the farm rather than just limited or single crop type (monoculture).
Hill	Refers to the risen bed where the crop is planted in a furrow irrigated field.
Honeydew	A sticky sugar rich waste excreted by feeding aphids or whiteflies. It can interfere with photosynthesis, affect fibre quality and cause problems with fibre processing.

HydroLOGIC	Irrigation management software available from CSIRO and the Australian Cotton CRC.
In-furrow insecticide	Insecticides applied in the seed furrow with the seed during planting. The insecticide may be applied as a granule or as a spray into the seed furrow.
Insecticide resistance	Where a pest develops resistance to an insecticide, the insecticide will no longer kill those individuals that are resistant. This usually results in poor control and may lead to failure of control with the insecticide in the worst cases. The resistant insects develop a mechanism for dealing with the insecticide, such as production of enzymes which break the insecticide down quickly before it kills the pest.
Insecticide Resistance Management Strategy (IRMS)	An industry regulated strategy that sets limits on which insecticides can be used, when they can be used and how many times they can be used. This helps prevent the development of insecticide resistance.
Larval parasitoids	A wasp that lays their egg on or in a larva and use the lifecycle of the larva in order to reproduce. Parasitoids usually cause the death of their host whereas parasites do not.
Leaf crumpling	Leaves that are wrinkled, cupped and smaller than normal. This can be caused by thrips.
Lint	Cotton fibres. These are elongated cells growing from the surface of the cotton seed coat. See also 'Bolls'.
Main stem node	A point on the main stem from which a new leaf grows. At these points there may also be fruiting or vegetative branches produced.
Management unit	An area on the farm that is managed in the same way i.e. same variety, sowing date, insect management.
NACB	The number of main stem N odes A bove the first position C racked B oll. This is an indication of the maturity of the plant and can be used in making decisions about the final for irrigation or defoliation..
Natural enemies	Predators and parasitoids of pests.
Natural mortality	The expected death rate of insects in the field mainly due to climatic and other environmental factors including natural enemies.
NAWF	The number of main stem N odes A bove the first position W hite F lower that is closest to the plant terminal.
Neutron probe	An instrument used to measure soil moisture.
Node	A leaf bearing joint of a stem, an important character for plant mapping in cotton where nodes refer to the leaves or abscised leaf scars on the main stem.
Nursery	A crop or vegetational habitat which attracts and sustains an insect (pest or beneficial) through multiple generations.
NutriLOGIC	Nitrogen fertiliser management software in CottonLOGIC or on the Australian Cotton CRC website.
NUTRIpak	An information resource for cotton nutrition, including critical levels for soil tests, and interactions between different nutrients.
Nymph	The immature stage of insects which looks like the adult but without wings. Eg. nymphs of mirids. Nymphs gradually acquire adult form through a series of moults and do not pass through a pupal stage. In contrast, 'larvae' are immature stages of insects, such as the <i>Helicoverpa</i> caterpillars, that look quite different to the adults, which in this case is a moth.
Okra leaf type	Cotton varieties with deeply lobed leaves that look very similar to the leaves on the Okra (<i>Abelmoschus esculentus</i>) plant, which is related to cotton and hibiscus.
OZCOT model	A cotton crop simulation model that will predict cotton growth, yield and maturity given basic weather, agronomic and varietal data.
Pest flaring	An increase in a pest population following a pesticide application intended to control another species. This usually occurs with species that have very fast life cycles such as spider mites, aphids or whitefly. It occurs following the use of broader spectrum insecticides which control the target pest but also reduce the numbers of predators and parasites. This allows these 'secondary' or non-target pests to increase unchecked, often reaching damaging levels and requiring control.
Peak Flowering	The period of crop development where the plant has the highest numbers of flowers opening per day.
Pest damage	Damage to the cotton plant caused by pests. This can be either damage to the growing terminals (known as tipping out), the leaves, or the fruit (including squares or bolls).

Pest resurgence	An increase in a pest population following a pesticide application intended to reduce it. This usually occurs because the insecticide has reduced the numbers of beneficials, which normally help control the pest, thereby allowing subsequent generations of the pest to increase without this source of control.
Pest threshold	The level of pest population at which a pesticide or other control measure is needed to prevent eventual economic loss to the crop. See also 'Damage threshold'.
Petiole	The stalk that attaches the leaf to the stem.
Phase 1	The period between planting and the start of flowering (one flower per metre).
Phase 2	The period between flowering to first open boll.
Phase 3	The period between first open boll to harvest.
Plant available water	The amount of water in the soil that can be extracted by plants, usually full point (when the soil can hold no more water) minus wilting point (point at which the plant can no longer extract sufficient water from the soil and begins to wilt).
Plant growth regulator	Chemicals which can be applied to the plant to reduce growth rate (see also 'Rank growth').
Plant mapping	A method used to record the fruiting dynamics of a cotton plant. This can be useful for understanding where the plant has held or is holding the most fruit in order to interpret the effects of factors that may affect fruit load such as pest damage, water stress, heat.
Plant stand	The number of established cotton plants per metre of row.
Post-emergent knockdown herbicide	A herbicide used to rapidly control weeds after they emerge.
Predator to pest ratio	A ratio used to incorporate the activity of the predatory insects into the pest management decisions. It is calculated as total number of predators per metre divided by the total number of <i>Helicoverpa</i> spp. eggs plus very small and small larvae per metre.
Premature cut-out	Premature cut-out is when the production of bolls exceeds the supply of carbohydrates too early in the crops development and therefore the production of new fruiting nodes stops. This results in a less than ideal boll load.
Pre-plant knockdown herbicide	A herbicide used to rapidly control weeds prior to planting.
Presence/absence	The binomial insect sampling technique that records the presence or absence of a pest rather than absolute numbers on plant terminals or leaves, depending on the pest species being sampled.
Prophylactic	Refers to regular insecticide sprays applied in anticipation of a potential pest problem. Spraying on a prophylactic basis runs the risk of spraying to prevent pest damage that would not have occurred anyway, thereby increasing costs, selection for insecticide resistance and the risk of causing secondary pest outbreaks due to reductions in predator and parasite numbers.
PSO	Petroleum Spray Oil – are petroleum derived oil commonly used to control insect pests such as <i>Helicoverpa</i> spp., mirids, mealy bugs, aphids, thrips, scales and mites. PSOs can also be used to deter egg lay of some pests such as <i>Helicoverpa</i> spp.
Pupae	Once larvae of <i>Helicoverpa</i> have progressed through the larval (caterpillar) stages they will move to the soil and burrow below the surface. Here they will change into a pupae (similar to a butterfly chrysalis). In this stage they undergo the change from a caterpillar to a moth.
Rank crop	A rank crop is usually very tall (long internode lengths) with excessive vegetative plant structures. This can be caused by a number of factors including excessive fertilizer use, pest damage and crop responses to ideal growing conditions especially hot weather. Rank crops can be difficult to spray and to harvest and may have delayed maturity or reduced yield. Seed company web sites detail methods to assess plant growth to test if a plant growth regulator might be needed to prevent such rank growth.
Ratoon cotton	A cotton crop in which the stalks are cut down after harvest, but the crown and rootstock are left in the ground to regrow the following season. For pest and disease reasons, this form of cropping is not used in Australia.
Refuge	This term is used to refer to crops grown specifically as a requirement of the Bollgard II® licence to produce <i>Bacillus thuringiensis</i> (Bt) susceptible <i>Helicoverpa</i> spp.
Rotation crops	Other crop types grown before or after the cotton is grown.
Secondary pests	Pests such as spider mites, aphids or whiteflies which do not usually become a problem unless their natural enemies (predators or parasites) are reduced in number by insecticides. See also 'Pest Flaring'.
Seed bed	A type of mound on which furrow irrigated cotton is grown.

Seed treatment	An insecticide / fungicide used to coat cotton seeds to offer a period of protection during germination and establishment against some ground dwelling pests eg. wireworm and some early foliage feeders such as thrips or aphids.
Selection pressure	The number of times insecticides from a particular chemical group are sprayed onto a cotton crop. Each of these spray events will control susceptible individuals, leaving behind those that are resistant. More selection events means that there is greater 'pressure' or chance of selecting a resistant population.
Soil water deficit	The difference between a full soil moisture profile and the current soil moisture level.
Square	Cotton flower bud.
Squaring nodes	A node at which a fruiting branch is produced, which is defined as a branch with a square which has a subtending leaf that is fully unfurled and on which all central veins are visible.
Standing stubble	Stalks from a crop that has been harvested or sprayed out and left to stand in the field.
Sucking pests	Pest, usually from the group of insects known as hemiptera or bugs which have piecing tubular mouthparts which they insert into plant parts to obtain nutrition. Key among these are green mirids, which feed on cotton terminals, and young squares and bolls. Some bugs inject toxins into the plant when they feed, which if bolls are fed on may cause seed damage and staining of lint.
Sweep net	A large cloth net (approximately 60 cm deep) attached to a round aluminum frame which is about 40 cm in diameter with a handle (1 m in length) used to sample insects.
Synthetic insecticides	Non-biological insecticides. They may be man made versions of natural insecticides (i.e. pyrethroids are synthetic, light stable versions of naturally occurring pyrethrum) or they may simply be man made molecules with insecticidal or miticidal (controls mites) activity. In this guideline we have used the term to encompass most insecticides with the exception of Bt sprays, virus sprays, food sprays and petroleum spray oils (PSOs).
Terminal	The growing tip of a cotton stem, particularly the main stem.
Tip damage	When the plant terminal has been damaged, also known as tipping out.
Top 5 retention	The percentage of first position fruit maintained on the top 5 fruiting branches.
Trap crop - last generation	A crop grown to concentrate <i>Helicoverpa</i> moths emerging late in the cotton season from the non-diapausing component of pupae from the last generation in autumn. These pupae are likely to be more abundant under conventional cotton and will have had intense insecticide resistance selection. The aim is to have these moths lay their eggs in the trap crop where the resulting pupae can be controlled by cultivation.
Trap crop – Spring	A crop grown to concentrate <i>Helicoverpa armigera</i> moths emerging from diapause, usually between September and October. These moths will establish the first generation of larvae in these crops, where they can be killed using biological insecticides (ie. virus sprays) or by cultivation to kill the resulting pupae.
Trap crop – Summer	A crop grown to draw <i>Helicoverpa armigera</i> away from a susceptible crop like cotton, and which can also produce large numbers of beneficial insects. The aim is to have these moths lay their eggs in the trap crop where the resulting larvae can be controlled using biological insecticides (i.e. virus) or the pupae controlled by cultivation.
True leaves	Any leaf produced after the cotyledons.
VAM	Vesicular Arbuscular Mycorrhiza: A partnership between soil borne fungi and most crop plants, including cotton (but not brassicas). Vesicular arbuscular mycorrhizal fungi colonise the roots of the plant without causing disease. The VAM fungi act as an extension of the root system and transfer extra nutrients, especially phosphorus, from the soil to the plant. In return the plant provides the fungi with sugars as a food source.
Vegetative growth	The roots, stems and leaves as distinct from the reproductive growth of flowers and bolls.
Visual sampling	Sampling insects in the field with the naked eye without the use of other equipment. See also 'Beat sheets', 'Sweep net' and 'D-vac'.
Water stress	When the demand for water to maintain plant function exceeds the amount available to the plant from the soil.
Waterlogging	When the plant roots endure a prolonged period under water, the lack of oxygen impairs water and nutrient uptake, both of which will have a direct effect on growth and yield.
WATERpak	An information resource for cotton water use and management.

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