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.

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

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.

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


Three banded ladybird

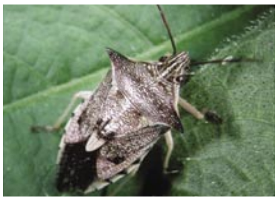

Transerve ladybird


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

Counting insects on a 'beat sheet'.

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.

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

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 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 effectivness 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. Recommence 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:

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).

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 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 pyrethoids, 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 and *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 (*Crocidosema 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:

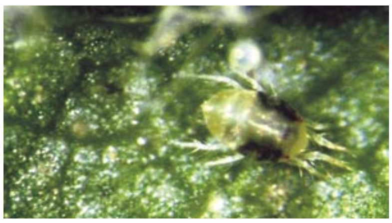
(a) Walk about 40 m into the field (early in the season it is advisable to sample near the field edges to see if significant influxes of mites have occurred),

(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 yeild 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 to10 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 liebknechti)

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 probelm 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 knows as the silverleaf whitefly. These two biotypes cannot be distinguished visually. Biochemical tests are required to separate the species.

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

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.

Silverleaf whitefly adults appear narrow when viewed from above.

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.

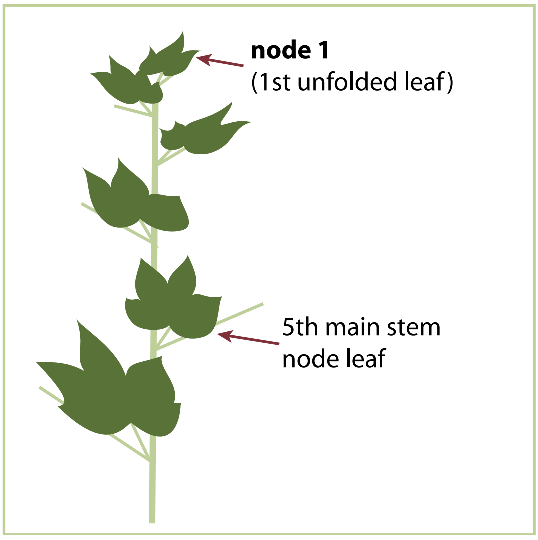


Figure 3.
Sample the fifth main stem node leaf.

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.

Counting the nymphs

- Sample for nymphs within a 3.88 cm^2 (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.

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	*Spodoptera exempta*
Lesser armyworm	*Spodoptera exigua*

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

Rough bollworm larva in a cotton boll.

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 1cm then leaf area reduction is often greater than 80%.

Young cotton damaged by thrips.

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.

Jassids

There are at least two species found in cotton, the vegetable leafhopper (*Austroasca viridigrisea*) and the cotton leafhopper (*Amrasca terraeregi-nae*). 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 Cental 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 infesta-tion 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 occassional 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.

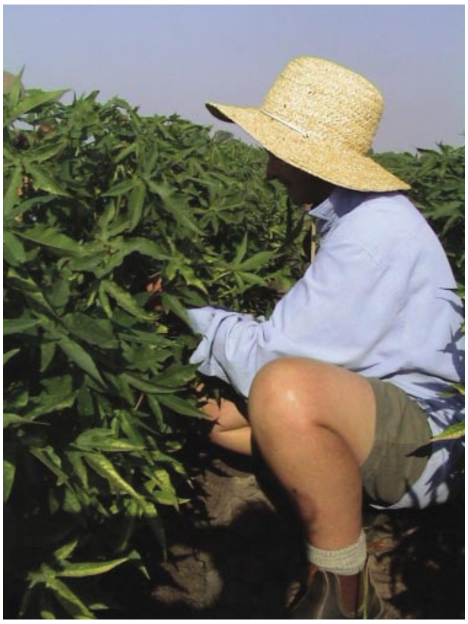
There is a range of tools available to assess fruit load. These include:

- 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.



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

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

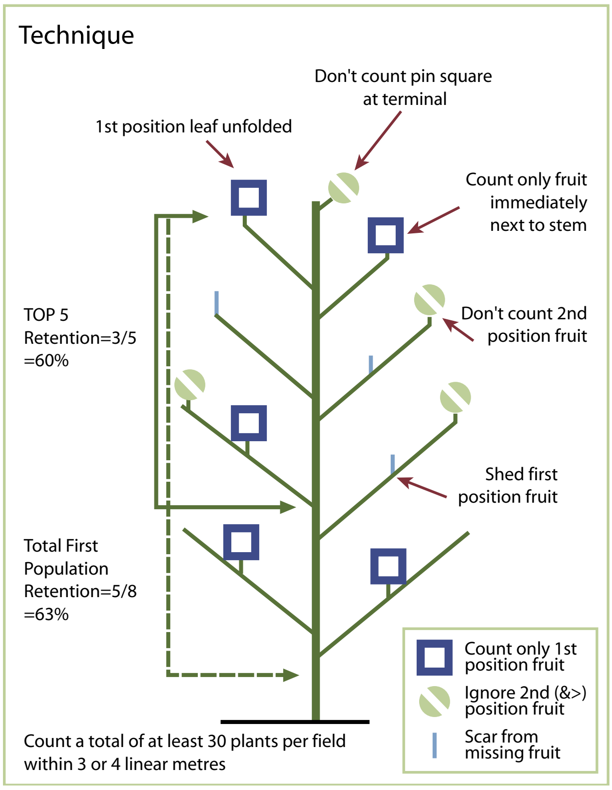


Figure 5.
A technique for checking fruit retention.

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 to14 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.

Yellow night stalker feeding on a green mirid adult.

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*'.

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

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.

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 theshold 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.

Multi-branched cotton caused by mirid feeding.

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 samplingfor 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 chemicials 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 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 then 21 days old, particularly those less then 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 nymphis 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	
	Presence/absence large nymphs of 30 discs sampled	Actual numbers/leaf disc (average)	Fewer than 3/leaf	At least 3-5/leaf
Whitefly Adult Levels	Less than 8 infested	Less than 0.5 average	Wait and re-sample in 3-7 days	Re-sample in 3 days OR apply IGR
	At least 8-12 infested	At least 0.5-1.0 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 disorted in shape.

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

***Helicoverpa*/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

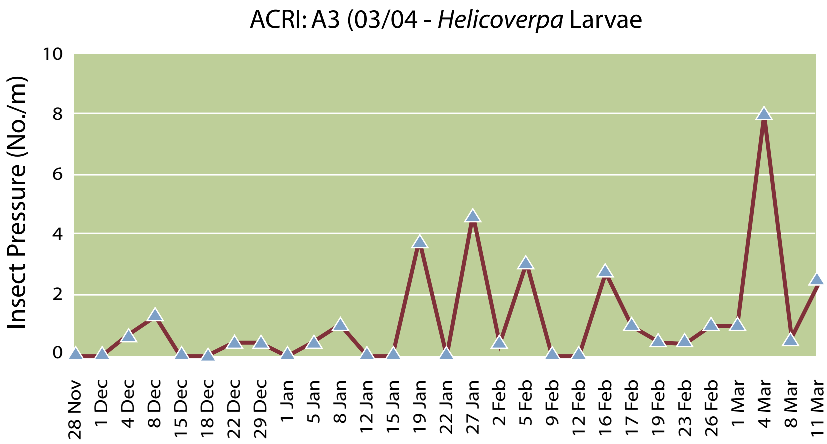
NB Thresholds for other pests and damage are as above

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		
Monitor plant damage: • Tip Damage • Fruit Retention	*Monitor pest densities:* • Caterpillars • Mirids and GVB • Mites • Aphids • Whitefly	*Monitor beneficials:* • 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		
Does crop need protection? *Consider:* I. Is it ahead of schedule and can tolerate some damage? II. Is poor retention due to pests or other factors?	*Are pests over, or close to the threshold?* *Consider:* Are pests increasing, static or decreasing? For pests such as mites, aphids and whitefly static or decreasing populations often indicate good predation levels.	*Are predator numbers or parasitism rates too low to control pests?* *Consider:* I. Proximity to sources of beneficials. II. Other pests present that beneficials may be controlling.
NO… Come back later and check again in two days		
YES… Choose an option		
ACTION		
Consider application issues: I. Wind, temperature, sensitive areas, Bt refuge regulations II. Impact of a spray on neighbours and nearby crops	*Check IRMS regulations* *Consider:* I. Available options II. Consecutive sprays (resistance in target and other pests) III. Application limits IV. Other pests present that may require control or be 'flared' i.e. mites, aphids, whitefly	*Check impact table* *Consider:* I. Impact on beneficials II. Potential to flare pests III. Using food sprays or biological insecticides or manipulating lucerne trap crops to restore the predator to prey ratio
EVALUATION		
Continue to monitor plant damage. • Is the plant on track? • If not, is it pest related or due to other factors.	*Resample* • 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)