NORTHERN

SEPTEMBER 2018

GRDC™ GROWNOTES™

GRDC™ GRAINS RESEARCH & DEVELOPMENT CORPORATION

CANOLA

SECTION 7

INSECT CONTROL

INTEGRATED PEST MANAGEMENT | EARTH MITES | LUCERNE FLEA | SLUGS | DIAMONDBACK MOTH | APHIDS | RUTHERGLEN BUG | HELICOVERPA | OTHER SOIL PESTS

MORE INFORMATION

Insect and mite control in field crops

Viral diseases in canola and winter pulses

Emerging insect pests

'Serial pests' wrap-up—lessons from 2014 and 2015 and some research updates

Canola disease update 2015 (NSW)

Insect pests of canola

Melina Miles QDAF discusses Rutherglen bug. GRDC Radio (Northern Update) 106: Safe seed storage strategy

Insect control

Insects that can pose a problem in canola in NSW include blue oat mites (*Penthaleus* spp.), redlegged earth mites (*Halotydeus destructor*), cutworms, diamondback moth, *Helicoverpa*, aphids Rutherglen bugs, slugs, European earwigs, Lucerne flea and wire worms.

The seed dressing Gaucho® (imidacloprid) protects emerging seedlings from redlegged earth mite, blue oat mite and aphids for ~3–4 weeks after sowing. Another seed dressing, Cosmos® (fipronil) protects seedlings from redlegged earth mites.

Viruses can also occur in canola, carried by aphids that suck sap from leaves, transferring the virus and causing yield loss and sometimes plant death. Protection against early aphid infestation in seedling canola may reduce the incidence of virus in the crop.

Gaucho® (imidacloprid) is the only seed dressing registered for control of aphids in emerging canola. Sowing canola into standing cereal stubble may help to reduce aphid numbers and hence virus infection. [1]

7.1 Integrated pest management

Pests are best managed using an integrated pest management (IPM) approach. Careful planning prior to sowing, followed by regular monitoring of crops after sowing, will ensure that potential problems are identified and, if necessary, treated early. Monitoring may involve techniques and aids such as sweep nets, a beat sheet or visual assessment.

Integrated pest management uses a range of control tactics to keep pest numbers below the level where they cause economic damage. It is primarily based on biological control of pests, by either encouraging natural enemies or release of biocontrols.

Other methods of control support these biological controls and can include:

- cultural methods such as farm hygiene, weed control, strategic cultivation (pupae busting), physical barriers, quarantine areas, different planting times, crop rotations, trap crops, use of attractants for beneficials or repellants for pests, and keeping plants healthy so they resist attack
- host plant resistance such as genetically resistant varieties or physical features that repel pests
- genetic control measures such as release of sterile male insects
- pheromones to confuse mating or aggregation
- use of microbial pesticides such as *Bacillus thuringiensis* (Bt), nuclear polyhedrosis virus (NPV) or *Metarhizium*.
- manipulation of micro-environmental conditions (e.g. planting density, row spacing, row orientation) to make them less suitable for pests or more suitable for beneficials
- use of chemicals as a last resort
- use of 'soft' chemicals or pest-specific chemicals in preference to broad-spectrum pesticides (especially early in the growing season when it is important to preserve beneficials).

1 L Serafin, J Holland, R Bambach, D McCaffery (2009) Canola: northern NSW planting guide. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0016/148300/canola-northern-NSW-planting-guide.pdf

Integrated pest management relies on monitoring the crop regularly, having pests and beneficial insects correctly identified and strategic control decisions made according to established damage thresholds. [2]

7.1.1 Area-wide management

Area-wide management (AWM) is IPM that operates over a broad region and attacks the pest when and where it is ecologically weakest, without regard to economic thresholds. It is a system currently used in managing resistance in *Helicoverpa armigera* in cotton. AWM coordinates farmers in implementing management strategies on their own farms to control local populations of *H. armigera* and prevent numbers building up later in the season. AWM strategies involve a detailed understanding of the biology and life cycle of the pest and of how the pest moves around in a region. Strategies can include coordinated timing of operations such as pupae busting, sowing and destroying of trap crops, and spraying of certain chemical types including 'soft' or biological insecticides. [3]

7.1.2 Biological control

Biological control can be defined as the use of natural enemies to control pest outbreaks. The pest is not usually eradicated, but brought down to levels where it does not cause economic damage. Success with biological control has been varied in many situations. Complete success, where the pests do not exceed the economic thresholds, has occurred in only 19% of cases. Many releases of biocontrol agents have had no significant effect. Success has been more common in long-term agro-ecosystems such as orchards and forests, where pest and natural enemy populations are more stable. In annual cropping systems, maintaining resources that favour the buildup of natural enemies, for example greater biodiversity of plants, retaining stubble and groundcover and limiting use of broad-spectrum insecticides, will help to keep pests in check. [4]

2 K Hertel, K Roberts, P Bowden (2013) Insect and mite control in field crops. NSW DPI Management Guide. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0005/284576/Insect-and-mite-control-in-field-crops-2013.pdf

3 K Hertel, K Roberts, P Bowden (2013) Insect and mite control in field crops. NSW DPI Management Guide. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0005/284576/Insect-and-mite-control-in-field-crops-2013.pdf

4 K Hertel, K Roberts, P Bowden (2013) Insect and mite control in field crops. NSW DPI Management Guide. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0005/284576/Insect-and-mite-control-in-field-crops-2013.pdf

Figure 1: *Beneficial insects can be help bring down insect pest incursions.*

Photo: Melina Miles, QDAF

Types of biological control

- Natural. An existing control agent is encouraged to control pests. This means avoiding the use of chemicals that may destroy these control agents.
- Single release (classical). The control agent is released with the aim of establishing it as a permanent part of the ecosystem. This is usually carried out for introduced pests.
- Multiple release. The control agent is not usually perfectly adapted to the environment (e.g. drought- or frost-intolerant) and so needs to be re-released. This may occur as a 'top-up' after unfavourable conditions, as a regular seasonal release, or as an inundative release where the control agent does not survive well. In this case, the agent is used as a 'living insecticide' that is released in large numbers to reduce pest numbers before it dies.

Biological control agents for insect pests can include:

- Predators. These actively capture their prey. Beetles, lacewings, bugs, flies, spiders and vertebrates are predators.
- Parasitoids. These are host-specific and need only one host to complete their life cycle. They lay eggs in their host and emerge after using the host as a food source. The host is nearly always killed. Parasitoids differ from parasites, which will coexist with the host. Parasitoids include wasps—e.g. the parasitic wasp, which will lay eggs inside lucerne aphids, white cabbage moth caterpillars and scarab larvae—and flies such as the tachinid fly.

- Pathogens. These include bacteria, viruses, fungi, protozoa and nematodes. A few of these organisms can enter and multiply rapidly within the host, e.g. *Metarhizium*, Bt, NPV.

Parasitoid and pathogen control agents are usually more successful than predators because they are more host-specific.

Trichogramma wasps

Trichogramma pretiosum wasps prey on the eggs of *Helicoverpa* spp., loopers, cabbage moths and others, and are suitable for use in minimally sprayed field crops, maize and vegetables, and for *Helicoverpa* in fruit crops. They are <0.5 mm in size and lay their eggs into moth eggs. The wasp larvae develop into a fully formed wasp inside the moth egg in the process killing the developing caterpillar. *Trichogramma* are supplied as parasitised moth eggs in capsules. These are distributed around the crop and can be applied with water. [5]

Nuclear polyhedrosis virus (NPV)

Insect viruses are naturally occurring, insect-specific pathogens that have been part of the environment for millions of years and play an important role in the natural control of insect populations. Insects consume the virus from the leaves. The virus then moves through the gut wall and invades the body of the insect, causing the insect to stop feeding and die within 5 days because of the breakdown of its internal organs. The body ruptures after death, releasing virus particles that infect other caterpillars. Gemstar® and Vivus Gold® are commercial products that control *Helicoverpa punctigera* and *H. armigera* with a liquid concentration of virus particles in cotton and selected crops. Typically, they provide 60–90% control of larvae. Both products fit best within an IPM program that uses natural enemies such as ladybeetles and parasites, but can be alternated with synthetic insecticides. As a biological insecticide, efficacy is dependent on environmental conditions for good performance. It needs to be ingested; therefore, coverage of the target area is essential. [6]

Bacillus thuringiensis

The Bt bacteria produce proteins that are characterised by their potency and specificity to certain species, most of which are agronomically important pests. Mixtures of protein crystals and spores have been sprayed in the same way as a chemical pesticide for many years in horticultural industries, but with variable success in broadacre field crops, Full-Bac® WDG being a notable exception. Full-Bac® WDG is a dry flowable, more suited to application by boom spray than previous formulations. The caterpillar ingests the protein, which then attacks the gut wall, causing holes, and the insect stops feeding. The bacterial spores contained in the protein then leak through the gut wall and cause bacterial infection. The insect will die, either from this bacterial infection or from starvation. This is the process that makes the Bt protein highly specific and environmentally desirable. Insertion of the Bt gene into cotton plants has taken many years to develop, and breeding is ongoing of plants that express higher levels of the Bt toxin. Resistance to Bt is being carefully monitored and controlled with the development of management programs and new research on multiple insect-resistance genes. Novel strains of Bt are being isolated for a wide range of pest families, including beetles, flies and locusts. [7]

MORE INFORMATION

Integrated Pest Management Fact Sheet

5 K Hertel, K Roberts, P Bowden (2013) Insect and mite control in field crops. NSW DPI Management Guide. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0005/284576/Insect-and-mite-control-in-field-crops-2013.pdf

6 K Hertel, K Roberts, P Bowden (2013) Insect and mite control in field crops. NSW DPI Management Guide. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0005/284576/Insect-and-mite-control-in-field-crops-2013.pdf

7 K Hertel, K Roberts, P Bowden (2013) Insect and mite control in field crops. NSW DPI Management Guide. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0005/284576/Insect-and-mite-control-in-field-crops-2013.pdf

7.2 Earth mites

Earth mites are the major pests of seedling canola, especially in central and southern NSW. Damage can be caused by redlegged earth mites and blue oat mites, which often occur in mixed populations. *Bryobia* mites are an increasing problem in some areas. A good mite-control program starts with a population reduction treatment the previous spring (Table 1). Learn to identify these three species of mites to ensure that the correct insecticide and rate is applied to the correct species.

Bare earth treatments. Germinating and establishing crops can be protected by:

- boom spraying the soil surface of previous pasture or high-risk paddocks with a residual insecticide immediately after sowing
- perimeter spraying bare ground in low-risk paddocks, not forgetting to spray around trees, rocky outcrops and dams, and along water flow-lines.

If you are unsure of the level of risk from mites, spray the whole paddock. Three bare earth sprays are registered that will give several weeks of residual protection. Bifenthrin is registered for redlegged earth mite, blue oat mite and *Bryobia* mites, but application rates vary according to the mite species being targeted. Alpha-cypermethrin will control redlegged earth mite, while methidathion is registered for both redlegged earth mite and blue oat mite.

Seed dressings. Imidacloprid (e.g. Gaucho®) and Poncho® Plus (clothianidin + imidacloprid) are registered for use on canola seed for protection against redlegged earth mite, blue oat mite and aphids. Poncho® Plus is also registered to control lucerne flea, wireworm and cutworm. A third seed dressing, Cruiser® Opti (thiamethoxam + lambda-cyhalothrin) is registered for suppression of redlegged earth mite and lucerne flea. These seed dressings will protect emerging seedlings for 3–5 weeks after sowing. Use treated seed following a pasture phase if a well-timed spring spray of insecticide has been applied. Apply a bare-earth border spray where untreated pastures border the canola crop. Seed companies can supply seed pre-treated with imidacloprid, Poncho® Plus and Cruiser® Opti. Cosmos® Insecticidal Seed Treatment (fipronil) is also registered for control of redlegged earth mite in canola. Even where a seed-dressing or bare-earth treatment has been used, it is advisable to check seedling canola regularly for mite damage. [8]

Redlegged earth mite and blue oat mite (*Penthaleus major*) are two soil-dwelling mites that damage crops in autumn, winter and spring. They are primarily pests of seedlings but can also seriously injure older plants. Winter crops at establishment may be severely damaged, particularly if growth during and following emergence is slow. Damaged plants die or remain stunted and weak. Sometimes seedlings are killed before they emerge. Both mites prefer light, sandy or loamy, well-drained soils and often occur together in crops on the Tablelands, Slopes and Plains of NSW. [9]

MORE INFORMATION

Nutrient management may double as pest control

8 P Matthews, D McCaffery, L Jenkins (2016) Winter crop variety sowing guide 2016. NSW DPI Management Guide. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0011/272945/winter-crop-variety-sowing-guide-2016.pdf

9 K Hertel, K Roberts, P Bowden (2013) Insect and mite control in field crops. NSW DPI Management Guide. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0005/284576/Insect-and-mite-control-in-field-crops-2013.pdf

Table 1: *Recommended control strategies for earth mites.* [10]

Pre-season (previous spring–summer)
Assess risk
High-risk situations:
History of high mite pressure Pasture going into crop Susceptible crop being planted (e.g. canola, pasture, lucerne) Seasonal forecast is for dry or cool, wet conditions that slow crop growth
Actions if risk is high:
Ensure accurate identification of species Use Timerite® (redlegged earth mites only) Heavily graze pastures in early-mid spring
Pre-sowing
Actions if risk is high:
Use an insecticide seed dressing on susceptible crops
Plan to monitor more frequently until crop established
Use higher sowing rate to compensate for seedling loss
Consider scheduling a post-emergent insecticide treatment
Actions if risk is low:
Avoid insecticide seed dressings (esp. cereal and pulse crops)
Plan to monitor until crop establishment
Emergence
Monitor susceptible crops through to establishment using direct visual searches
Be aware of edge effects; mites move in from weeds around paddock edges
Actions if spraying:
Ensure accurate identification of species before deciding on chemical
Consider border sprays
Spray prior to the production of winter eggs to suppress populations and reduce risk in the following season
Follow threshold guidelines
Crop establishment
As the crop grows, it becomes less susceptible unless growth is slowed by dry or cool, wet conditions

Feeding

Mites feed by rasping the surface of the cotyledons and leaves and by sucking up the sap. Feeding is normally from late afternoon until early morning, but continues through the day in calm, cloudy weather. Mites are very active and, if disturbed on a plant, will drop or descend to the ground and disperse to find shelter. Redlegged earth mites usually remain clustered together on the soil or on parts of the leaves during the day. Blue oat mites generally hide by day in the soil beneath damaged plants or under plant debris on the ground. [11]

10 P Umina (2014) Persistent pests; aphids, mites, millipedes and earwigs. GRDC Update Papers, 5 February 2014, http://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2014/02/Persistent-pests-aphids-mites-millipedes-and-earwigs

11 K Hertel, K Roberts, P Bowden (2013) Insect and mite control in field crops. NSW DPI Management Guide. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0005/284576/Insect-and-mite-control-in-field-crops-2013.pdf

7.2.1 Redlegged earth mite

The redlegged earth mite is mainly a pest on the Southern and Central Tablelands, Slopes and Plains. It is native to southern Africa and cape weed is its preferred host plant. Other hosts include prickly paddy melon, wild turnip, common sowthistle, Paterson's curse and chickweed (weeds); and canola, lupins, field peas and linseed (field crops). Sometimes mites may move into young winter cereals from a fence line or adjoining pasture and cause damage along one or more of the crop edges.

Description

Adult mites are eight-legged and ~1 mm long with oval, flattened black bodies and pinkish-orange legs and mouthparts.

MORE INFORMATION

Persistent pests; aphids, mites, millipedes and earwigs

Seasonal development

Three overlapping generations usually occur between mid-autumn and spring, and adult populations are normally highest in May–June and September–October. Redlegged earth mites oversummer as unlaid, aestivating eggs in the dead bodies of spring-generation adult mites lying on or near the soil surface. The aestivating eggs are highly resistant to desiccation and usually do not begin to develop until late summer–early autumn. They hatch when favourable conditions of soil temperature and moisture occur in the following mid-autumn to early winter.

TIMERITE® for management of redlegged earth mite

TIMERITE® is an information package that provides individual farmers with the optimum spray date on their farm to control redlegged earth mites during spring. Developed by CSIRO and Australian Wool Innovation, TIMERITE® predicts the optimum date in spring to control redlegged earth mites, just after they have ceased laying normal winter eggs on pasture and just before diapause. (Diapause is when adult redlegged earth mites produce eggs that are retained in the body of the adult female and are therefore protected from the effects of insecticide applications.) The single, strategic spray has a two-fold effect, controlling redlegged earth mites in spring and decreasing the summer population that emerges in the following autumn. The package may form part of an integrated management strategy to control redlegged earth mites.

Close attention should be paid to individual pesticide labels when controlling earth mites. Application rates vary with situations, such as bare earth or post-crop–pasture emergence. Correct identification of earth mite species is essential. Registrations sometimes include redlegged earth mites only, not blue oat mites or *Bryobia* mites. Application rates may vary with earth mite species. READ THE LABEL.

MORE INFORMATION

TIMERITE information package

This strategic approach has little effect on non-target invertebrates, both pest and beneficial, during the following autumn. Farmers need to identify geographically the location to be sprayed. This can be done by a local feature, such as town or mountain, or the longitude and latitude of the area. This information is used to find the optimum date from the package. The spray date for each farm is the same date each year. For information, phone Australian Wool Innovation toll free on 1800 070 099 or visit the website www.timerite.com.au. [12]

7.2.2 Blue oat mite

Blue oat mites are often confused with redlegged earth mites. There are four recognised species of blue oat mites in Australia: *Penthaleus major*, *P. falcatus*, *P. minor* and *P. tectus*. Accurate identification of the species requires examination by an entomologist. The four species vary in their geographical distribution in Australia. With the exception of *P. minor*, all species have been found in NSW, in some instances in mixed populations. Damage to crops and pastures is incurred in the establishment phase. Host-plant preferences vary with the species, as do their life cycles and tolerances to various pesticides. Host plants include black thistle, chickweed, curled

12 K Hertel, K Roberts, P Bowden (2013) Insect and mite control in field crops. NSW DPI Management Guide. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0005/284576/Insect-and-mite-control-in-field-crops-2013.pdf

dock, dandelion, deadnettle, prickly lettuce, shepherds purse, variegated thistle and wild oat. Cultivated field-crop hosts include wheat, barley, oats, rye, canola, field peas, lupins and linseed.

Description

Adult mites have eight legs and are ~1 mm long with oval, rounded, dark brown to black bodies, bright red or pinkish red legs and mouthparts, and a red spot or streak towards the hind end of the back.

Seasonal development

Overlapping generations of the blue oat mite usually occur between mid-autumn and late spring. Blue oat mites oversummer as aestivating eggs laid in mid–late spring by the second-generation adults. These aestivating eggs are highly resistant to desiccation. They do not begin to develop until late summer–early autumn and they do not hatch until favourable conditions of temperature and moisture occur in the following mid-autumn to early winter. [13]

7.3 Lucerne flea

Lucerne flea is an occasional pest of establishing canola crops. The pest is identified by its action of jumping and hopping between plants rather than flying. It is present across a range of soil types in southern NSW. Early-sown crops are more at risk of attack. Frequent crop inspection from the time of emergence and early control measures are important because of the impact of seedling vigour on crop performance. Ensure that monitoring is sufficient to detect localised patches or 'hot spots'. Seek advice on management and spray strategies. [14]

7.4 Slugs

Slugs are a potential problem along the Northern, Central and Southern Slopes, and occasionally adjacent to rivers on the Western Plains. Slugs kill plants at the seedling and rosette stages and can leave large, bare-soil areas. Slugs are favoured by wet springs and summers, where abundant growth and damp conditions provide an ideal habitat. This allows slugs to breed and survive into autumn and winter, when they attack newly sown crops.

Canola sown into dense stubble or adjacent to grassy fence lines, creek banks or damp areas is at greatest risk because these areas provide an ideal habitat for slugs to survive over summer. Heavy, cracking soils provide additional hiding places for slugs. Closely monitor crops at risk for 6–8 weeks after sowing, so that any infestation can be treated with slug pellets containing metaldehyde. [15]

MORE INFORMATION

Diamondback moth in canola. Fact Sheet

7.5 Diamondback moth

Diamondback moth has been observed in canola crops for many years in NSW. The summer of 2001–02 favoured their build-up and they became a serious pest in the drought of 2002. Few, if any, crops have required spraying since, despite major drought in 2006 and 2009. Caterpillars of diamondback moth do most damage when large numbers are present in seedling crops or when they move from leaves to graze developing pods during crop ripening. Diamondback moth has developed resistance to a range of insecticides. Future management will involve regular monitoring and careful selection of control methods. [16]

---

13 K Hertel, K Roberts, P Bowden (2013) Insect and mite control in field crops. NSW DPI Management Guide. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0005/284576/Insect-and-mite-control-in-field-crops-2013.pdf

14 P Matthews, D McCaffery, L Jenkins (2016) Winter crop variety sowing guide 2016. NSW DPI Management Guide. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0011/272945/winter-crop-variety-sowing-guide-2016.pdf

15 P Matthews, D McCaffery, L Jenkins (2016) Winter crop variety sowing guide 2016. NSW DPI Management Guide. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0011/272945/winter-crop-variety-sowing-guide-2016.pdf

16 P Matthews, D McCaffery, L Jenkins (2016) Winter crop variety sowing guide 2016. NSW DPI Management Guide. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0011/272945/winter-crop-variety-sowing-guide-2016.pdf

Figure 2: *Management of diamondback moth involves regular monitoring and careful selection of control methods.*

7.6 Aphids

Aphid flights can occur in autumn and winter in some years and can infest young canola crops. Crops may need to be treated with insecticide to prevent transmission of virus diseases, but also to reduce seedling damage and the risk of spring infestations. The green peach aphid is the major vector of BWYV, which caused some crop damage in southern and central NSW in 2014. Seed treated with imidacloprid (e.g. Gaucho®), Poncho® Plus and Cruiser® Opti will protect seedling canola for up to 5 weeks. This is especially important in seasons and at sites where early infestation with aphids occurs. [17]

Green peach aphid has developed resistance to the synthetic pyrethroid, carbamate and organophosphate groups of insecticides. Transform™ (sulfoxaflor) is a new selective insecticide for control of early-season infestations of green peach aphid. [18]

Aphids can also infest crops in the spring, especially in years of moisture stress. Large populations of aphids are more evident and potentially damaging in dry seasons. Monitoring for beneficial insects is important, because control may not be justified in some cases. If control is warranted, careful selection of an insecticide is essential to ensure that damage is not caused to nearby beehives or to beneficial insects within the crop. Ensure that the harvest-withholding period (WHP) of the insecticide is adhered to. Seek advice on thresholds and product registrations or permits before spraying.

MORE INFORMATION

Reducing aphid and virus risk

Resistance management strategy for the green peach aphid in Australian grains

Unified attack on pests and diseases

Insect pests—resistance, virus vectors and lessons from 2014

Crop aphids. Back Pocket Guide

Ground Cover Radio 116: Aphids in canola it pays to wait

Melina Miles QDAF discusses aphid control. GRDC Radio (Northern Update) 084: Aphid control re-think

17 P Matthews, D McCaffery, L Jenkins (2016) Winter crop variety sowing guide 2016. NSW DPI Management Guide. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0011/272945/winter-crop-variety-sowing-guide-2016.pdf

18 P Matthews, D McCaffery, L Jenkins (2016) Winter crop variety sowing guide 2016. NSW DPI Management Guide. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0011/272945/winter-crop-variety-sowing-guide-2016.pdf

Figure 3: *Aphids can transmit damaging viruses to canola crops.*

Photo: Melina Miles, QDAF

IN FOCUS

7.6.1 Recent GRDC-funded research

Take home message

Compensatory capacity of canola supports the use of less conservative aphid thresholds and increased consideration of natural enemies in controlling outbreaks.

Outcomes and recommendations—management of aphids in canola

1. Simulated damage through the removal of raceme terminals by cutting provides an adequate way of simulating aphid damage.
2. Trials conducted on the Darling Downs in 2013 and the Liverpool Plains in 2014 show identical trends in terms of crop compensation for simulated aphid damage. Consequently, we have some confidence that the conclusions drawn from these trials will have application to canola crops across a wide range of northern region growing conditions.
3. Removal of the flowering portion of podding racemes (23 days post first flower) did not affect yield, probably because the flowers removed would not have set harvestable pods.
4. Simulated damage to the raceme during the first weeks of flowering had only minimal impact on final yield, except where extreme damage was enacted (66% of, or the entire, raceme removed on every plant).

5. Compensatory capacity and minimal maturity delay following damage to racemes suggests greater opportunity to harness the benefit of natural enemies in controlling aphid outbreaks.

Aphid populations are extremely difficult to work with in the field. Manipulating densities, frequency and persistence of infestations are major constraints to achieving trial outcomes. Therefore, simulated aphid damage is the only viable way to apply consistent treatments across a replicated trial (Figure 4).

The purpose of the simulated aphid trials reported here was to assess the impact of differing levels of damage at different stages of crop development. The trials were designed to evaluate the compensatory capacity of canola to recover yield if damaged by aphids at different stages of crop development. Understanding when canola is most susceptible to aphid-type damage is the first step in determining the need for, and timing of, aphid control in canola.

Experiments 1–3 were conducted on the Darling Downs with dryland canola (43Y85).

Figure 4: *The two methods of inflicting simulated aphid damage. Bagging racemes to prevent normal development, used in 2013 only (left), and cutting racemes (right).*

Experiment 1. Bagging canola racemes

Canola raceme development was inhibited by placing bags over the developing main stem raceme to limit development. Three treatments and an untreated control were applied to randomly assigned plots; the primary raceme on all plants was bagged at first flower (treatment 1), 7 days later (treatment 2) and 14 days after first flower (treatment 3), or not bagged (control).

Analysis of grain yields showed that all of the bagged treatments yielded significantly less than the control but were not different from each other regardless of treatment timing (Table 2). The maturity of the plots at the end of the season was very similar to the control, possibly 3–4 days behind in terms of the rate of plant senescence. This result shows that the later flowers (those prevented from developing at +7 and +14 days) contributed little to the final yield, or that soil moisture limited continued growth or compensation in the treated plots.

Table 2: *Plot yields (g/plot) from Experiment 1, Darling Downs, where canola raceme development was limited by bagging at 7-day intervals from first flower.*

Treatment	Yield	SE
Control (unbagged)	1293a	63
Treatment 1.Raceme covered at first flower	988b	18
Treatment 2. Raceme covered at first flower +7 days	989b	62
Treatment 3. Raceme covered at first flower +14 days	949b	25

SE, Standard error. Means followed by the same letter are not significantly different at P = 0.05, where l.s.d. = 146 g. Difference between control and treatments significant (P < 0.002

Experiment 2. Increasing severity of cutting flowering racemes

The experiment was laid out in the same canola field as used for Experiment 1 to test the effects of damage at different intensities during early flowering. Four treatments were applied with increasing severity at 14 days post first flower, and there was an undamaged control (Table 3).

The damage applied to the plots at 14 days post first flower resulted in a significant yield loss for only the most damaging treatment, in which all developing raceme terminals were cut (top 7 nodes) (Table 3).

The rate of crop senescence was marginally delayed in the two most severe cutting treatments. Crop maturity of treated plots was delayed by ~3–4 days compared with the control.

Table 3: *Plot yields (g/plot) from Experiment 2, Darling Downs, where canola racemes were cut with increasing severity at 14 days post first flower.*

Treatment	Yield	SE
Control (undamaged)	1137a	108
Terminal of raceme cut	1173a	98
Top 3 nodes of raceme cut	1133a	65
Top 5 nodes of raceme cut	1102a	89
Top 7 nodes of raceme cut (nearly all)	841b	111

SE, Standard error. Means followed by the same letter are not significantly different at P = 0.05, where l.s.d. = 242 g. Difference between most severe treatment and other treatments significant (P < 0.005

Experiment 3. Damage to flowering racemes at late flowering-pod filling

In this experiment, just the flowering portion of filling racemes was removed at 23 days after first flower. There were three treated plots, in which the terminals of 10%, 50% or 90% of the racemes in the plot were damaged, and an undamaged control (Table 4). These treatments were designed to simulate the late terminal infestations commonly observed in late spring.

This damage to the flowering terminals during late flowering–podset had no significant impact on grain yield (Table 4). There was no delay in crop maturity and no significant difference in the rate of crop senescence between treatments.

Table 4: *Plot yields (g/plot) from Experiment 3, Darling Downs, where increasing proportions of racemes were damaged during late flowering–pod set.*

Treatment	Yield	SE
Control	1246	70
10% of racemes terminal removed	1159	37
50% of racemes terminal removed	1193	65
90% of racemes terminal removed	1204	89

SE, Standard error. There were no significant treatment differences (P > 0.05

Experiment 4. Liverpool Plains 2014

The effects of simulated damage on flowering canola were further tested in a second series of experiments at eight sites near Spring Ridge on the Liverpool Plains, NSW. Three treatments were applied (one-third and two-thirds of raceme flowers and whole of raceme removed), with an undamaged control at each site.

The damage was inflicted during the early stages of flowering (within 7–10 days of first flower) at five sites during early August and at three sites where flowering began during late August. An analysis of oil quality and seed characteristics remains to be completed, but harvested grain weights indicated again that main treatment to suffer significant damage was where the entire raceme was removed, leaving plants to regrow completely new flowering structures (Figures 5, 6, 7).

The rate of crop senescence was marginally delayed in the more severe cutting treatments, being 3–5 days behind the undamaged controls, even in the most severe cutting treatment.

Treatment where two-thirds of raceme flowers were removed (top), and 15 days later showing compensation (bottom).

Treatment where entire raceme is removed (top), and 15 days later showing compensation (bottom).

Figure 5: *Images of treatments at time of treatment and compensation by the crops 15 days later, near Spring Ridge, Liverpool Plains.*

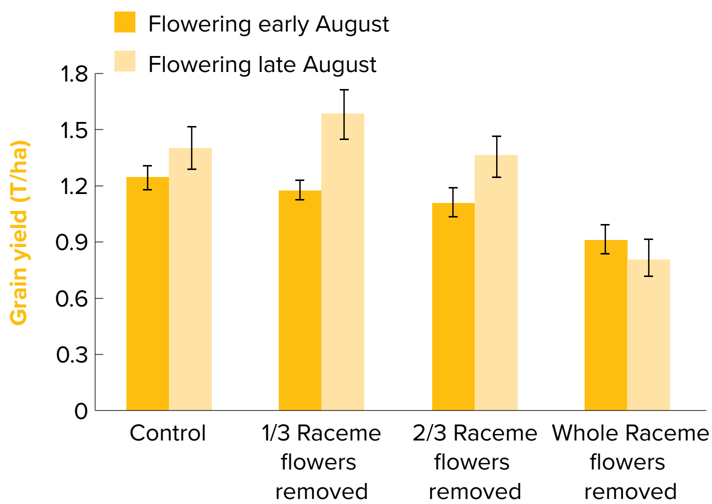

Figure 6: *Mean canola yield for treatments applied soon after first flower during early August (five sites) and late August (three sites), near Spring Ridge, Liverpool Plains. Capped lines denote treatment standard error.*

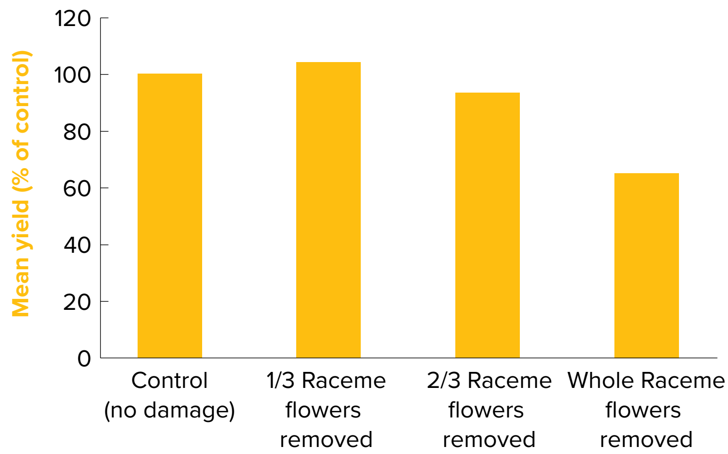

Figure 7: *Aggregate treatment yields across all sites near Spring Ridge, Liverpool Plains, expressed as a percentage of the control.*

Conclusions and observations

The bagging of canola racemes during early flowering had a significant effect on yield potential (Experiment 1). By contrast, cutting the racemes (Experiment 2) had an impact on yield only for the most severely damaged treatment that had all raceme terminals from the top seven nodes disrupted (essentially all flowering racemes on the plants). The yield of the other treatments was not significantly affected, although the second most severe treatment did tend towards a lower yield.

The difference in crop response to bagging and cutting is probably caused by the plants continuing to develop racemes under the bag for a significant part of the flowering period rather than deploying assimilates to newer, compensatory racemes from lower down in the plant canopy. This contrasts with the plants where damage was inflicted by cutting, in which compensatory growth of remaining racemes or the initiation of new racemes from adjacent nodes occurred rapidly after damage was inflicted.

Late damage to flowering–podding racemes did not have any effect on crop yield. The flowers removed from the racemes at this stage of flowering would not have set viable pods, so their loss did not have an impact on yield.

Although the physical damage inflicted during these experiments is different from that expected during an aphid infestation, the results suggest that canola has an excellent capacity to compensate for crop damage and that current spray thresholds of ~10% raceme infestation may be more stringent than necessary.

The capacity for compensation and the regular presence of effective natural enemies of aphids in canola provide an excellent opportunity to rely on, and allow time for, biological control by aphid parasitoids and ladybirds during the first weeks of flowering. A delay in enacting a spray decision at the 10% infestation level could be considered to hold low risk and would allow time for biological control. If natural enemies were ineffective, spraying on an increasing level of infestation to the 20–25% level would be unlikely to result in irrecoverable crop damage. Similarly, late infestations of aphids are unlikely to pose a damage threat to canola because the associated raceme disruption mainly affects flowers that contribute little to final yield.

MORE INFORMATION

Insect management in faba beans and canola recent research

Further study is planned for 2015 to determine a better linkage between the simulated damage in these experiments and actual damage caused by aphids in highly controlled, small-plot experiments, so that a definitive recommendation can be made on aphid thresholds. In the interim, the work suggests that the use of the 10% raceme infestation as a spray threshold is very conservative. [19]

7.7 Rutherglen bug

Rutherglen bug (RGB) is present in canola crops as they are filling pods and maturing. The RGB lay eggs in the soil (up to 400 eggs per female), resulting in an explosion of nymphs in 2–4 weeks after the adult RGB are seen. Although not typical, nymphs will move up the plants and onto the pods. More commonly they remain on the soil and base of plants. Where canola is windrowed, the nymphs may move to shelter under the windrows and feed on the shed seed. The risk of RGB damage to seed through direct feeding is not well understood and warrants research. Overseas research suggests that the risk of impact on grain (oil content/quality) decreases as the crop matures and dries down. [20]

7.8 *Helicoverpa*

Helicoverpa (also known as heliothis) caterpillars are an occasional pest of canola in southern NSW and may require control measures if they are present in large numbers. In central and northern NSW, they are a more frequent pest. Because of the seasonal variation in incidence and timing of infestation relative to crop growth stage, growers should seek advice and check the harvest WHP of the chosen insecticide before deciding to spray.

Corn earworm (*Helicoverpa armigera*) and native budworm (*H. punctigera*) are the two pest species of field crops and may be present from mid-September onward. Corn earworm is likely to predominate between February and May.

19 M Miles, P Grundy, A Quade, R Lloyd (2015) Insect management in in fababeans and canola recent research. GRDC Update Papers 25 February 2015, http://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2015/02/Insect-management-in-fababeans-and-canola-recent-research

20 M Miles, A Quade, R Lloyd, P Grundy (2016) Recent insect pest management research findings and the application of results in the field. GRDC Update Papers 23 February 2016, https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/02/Recent-insect-pest-management-research-findings-and-the-application-of-results-in-the-field

Figure 8: Helicoverpa armigera.

Photo: QDAF

Weekly trap catch data for *H. punctigera* and *H. armigera* from locations across all states can now be viewed online. The adjustable bar below the map allows selection of a time period (1 wk, 2 wks, 1 mth, etc). https://jamesmaino.shinyapps.io/MothTrapVis/

Seasonal biology

There are generally three or four overlapping generations of caterpillars of the native budworm and corn earworm between September and May in southern NSW, and four or five in northern NSW. [21]

Scout crops regularly

The amount of damage caused by native budworm and corn earworm varies considerably from year to year. Moth activity alone cannot be taken as a guide for spraying. In some years when moths are common, egg and caterpillar numbers are often limited by adverse cool or cold, wet weather, parasitoids, predators and diseases, and damage may be restricted or insignificant. In other years, a relatively small moth population may produce many caterpillars and cause significant damage. Periodic outbreaks of caterpillars of both species in summer are often associated with heavy rainfall. Check crops at least a week after heavy rainfall. Look for moths, eggs and very small caterpillars, and treat if necessary. Spraying thresholds are unlikely ever to be more than guidelines for timing sprays. Examine crops at least twice a week during the various danger periods.

Before deciding to spray, consider the following:

- likely extent and severity of the infestation
- ability of the crop to tolerate caterpillar damage without any significant loss or to replace leaves or fruiting parts lost to the caterpillars
- value or likely loss if the crop is left untreated
- cost of treatment. [22]

21 K Hertel, K Roberts, P Bowden (2013) Insect and mite control in field crops. NSW DPI Management Guide. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0005/284576/Insect-and-mite-control-in-field-crops-2013.pdf

22 K Hertel, K Roberts, P Bowden (2013) Insect and mite control in field crops. NSW DPI Management Guide. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0005/284576/Insect-and-mite-control-in-field-crops-2013.pdf

Spray eggs and very small caterpillars, particularly of the corn earworm

Corn earworm has developed resistance to many of the pesticides used against it, but the eggs and very small caterpillars (up to 5 mm long) can still be killed if the correct rate of pesticide is used. However, larger caterpillars are not likely to be controlled. [23]

7.8.1 Corn earworm

In NSW, corn earworm is largely restricted to within ~150 km of the coast and to inland, irrigated summer-crop production areas. Unlike native budworm, it mainly overwinters locally as pupae in the soil. These pupae are the source of the moths that produce the spring and summer generations of caterpillars. The moths are not strong fliers but can be carried for long distances by wind. They usually initiate infestations locally—within ~100 km of where they developed as caterpillars. During summer, corn earworm moths may be carried by wind into southern NSW from areas to the north (or into northern NSW from southern Queensland) and initiate infestations in crops over a wide area. Pheromone traps or lights do not always attract moths. There may be no forewarning of a probable outbreak of corn earworm unless growers regularly inspect crops to detect moths.

7.8.2 Native budworm

Native budworm is widely distributed throughout mainland Australia and during winter breeds in semi-arid parts of Western Australia, South Australia and south-west Queensland. These vast inland areas are the sources of the moths that produce the spring generation of caterpillars in NSW (local overwintering pupae in the ground are of little concern). The moths are strong fliers and may be carried for very long distances by wind, to initiate infestations in localities far from where they developed as caterpillars.

7.8.3 Descriptions of corn earworm and native budworm eggs and caterpillars

Eggs

Newly laid eggs are white or yellowish white, dome-shaped, flattened at the base, ribbed and 0.5 mm in diameter. Not all eggs are fertile. Fertile and infertile eggs are laid at the same time, and sometimes all eggs laid are infertile. Fertile eggs change to greenish yellow with an irregular brown or reddish brown ring around the middle. Before hatching, the blackish head and grey body of the caterpillar shows through. They hatch in 3–5 days in warm weather and 6–16 days in cooler weather. Infertile eggs appear cylindrical within ~12 h of being laid and then shrivel to a pyramid shape.

Caterpillars

Newly hatched caterpillars are 1–1.5 mm long with dark heads and dark-spotted white bodies. Young caterpillars up to about 15 mm long have dark heads and pale yellow, greenish or brownish bodies with conspicuous upper body hairs in dark bases and, often, narrow dark stripes down the back and along each side. Older caterpillars up to 50 mm long vary greatly in colour from yellow to almost black, often have a broad pale stripe along each side, and their upper body hairs are usually on raised processes.

Egg laying

Egg laying is usually confined to the period from flower bud formation until flowering ends. When moths are exceptionally abundant, infestation can be expected before flowering commences. Eggs are laid, usually singly, on the upper parts of plants—

23 K Hertel, K Roberts, P Bowden (2013) Insect and mite control in field crops. NSW DPI Management Guide. NSW Department of Primary Industries. http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0005/284576/Insect-and-mite-control-in-field-crops-2013.pdf

vegetative or floral growing points, young tender leaves, stems and flower buds, flowers and fruits. The moths prefer the more advanced and succulent portions of crops for egg laying and usually avoid poorly grown areas. Eggs may not be obvious to the untrained eye because of their minuteness. However, moderate to heavy egg lays should be obvious to trained observers. [24]

7.8.4 Resistance-management strategy

Corn earworm has developed resistance to the pyrethroid chemical group (e.g. Decis Options®, Karate®), and the carbamate group (e.g. Lannate® L). However, very small caterpillars (up to 5 mm long) can still be controlled with these and other pesticides. New chemical groups are now available for the control of corn earworm, but often control is slower.

Corn earworm caterpillars are found in all irrigation areas. Their main activity is in summer and autumn. Local overwintering pupae are the main source of new infestations each year.

The following strategies are recommended across all cotton and grain industries:

1. Destroying the overwintering pesticide resistant pupae

- Corn earworm overwinters as pupae in the soil under crop stubble and moths emerge from the pupae in the following spring.
- Insecticide resistance is carried over between seasons in the pupae.
- Where large *Helicoverpa* caterpillars are present in crops during March, there is a risk of carryover of resistance to the next season. Cultivate these paddocks to destroy the pupae as soon after harvest as possible and complete by the end of August.
- Check no-till, late-season crops for pupae. Consider busting if detectable numbers (1 pupa/10 m) are present.
- Ensure that cultivation creates full disturbance to at least 10 cm deep as follows: on hills, to 10 cm each side of the plant line; on beds, right across the bed to 20 cm beyond the outside rows; on the flat, the whole area.

2. Scout crops regularly

- Monitor crops twice weekly during danger periods to detect eggs and very small (up to 5 mm long) caterpillars. Infestations are often associated with heavy rainfall.
- Use advisors for monitoring, because moth activity alone cannot be taken as a guide for spraying and eggs are not readily seen with the naked eye—they are small and often hidden.
- Pay closer attention to late season *H. armigera* activity on susceptible crops (cotton, summer pulses, late sorghum and late sunflowers).

3. Spray eggs and very small caterpillars

- Spray to control the eggs and very small (up to 5 mm long) caterpillars. The chemical must make contact with the eggs and caterpillars.
- Egg laying is usually confined to the period from flower bud formation until flowering ends. At 25°C, it takes 8–10 days from egg lay to 5 mm long caterpillars. [25]

4. Pesticide management

- Conserve beneficials by using the most selective pesticide available, including appropriate use of NPV products (e.g. Vivus Gold®).
- Monitor natural enemies and be aware that their activity varies depending on crop types (e.g. they are not very active in chickpeas).

24 K Hertel, K Roberts, P Bowden (2013) Insect and mite control in field crops. NSW DPI Management Guide. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0005/284576/Insect-and-mite-control-in-field-crops-2013.pdf

25 K Hertel, K Roberts, P Bowden (2013) Insect and mite control in field crops. NSW DPI Management Guide. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0005/284576/Insect-and-mite-control-in-field-crops-2013.pdf

- Delay the use of disruptive pesticides (pyrethroids, carbamates and organophosphates) in all crops for as long as possible. Review spraying thresholds as a guide to spray timing.
- Where *Helicoverpa* populations are present and above threshold, control them within the limits of insect resistance and available registrations. Ensure that spray applications are accurate and timely. Do not spray in the heat of the day. Use an NPV product (e.g. Vivus Gold®) where appropriate to reduce selection for insecticide resistance, but caterpillars must be small (5–10 mm), the product must be applied in the evening, and there should be no rainfall within 24 h. Apply NPV as high-volume spray (with a wetting agent) at 30 L/ha aerially and 100 L/ha by a ground-rig.
- Rotate chemical groups when spraying crops. If more than one spray is needed, use a pesticide from each of the three available groups (organochlorines, synthetic pyrethroids and carbamates) in rotation. Never apply two consecutive sprays from the same chemical group. For example, if a pyrethroid is used to control sorghum midge do not use a pyrethroid to control *Helicoverpa*.
- Remove all farm vegetation likely to encourage the breeding of insect pests. For example, destroy failed or abandoned crops by cultivation or by using herbicides immediately the decision to abandon has been made. Avoid sowing commercial chickpea crops after June. Late flowering crops are a known host for the next generation of *H. armigera*.
- Use ovicide rates of pesticides only where crops are monitored regularly for eggs and larvae and where eggs are targeted.
- Use larvicide (highest) rates where there is no regular crop monitoring and where larvae are targeted. Use information from pheromone traps.
- During August, September and October, pheromone traps will be in place in some districts. Information gained will indicate whether more detailed sampling of spring host crops (e.g. winter cereals, pulses and weeds) is necessary.
- Advisers or growers wanting assurance on the numbers of corn earworm moths laying eggs in their crops should catch moths in a sweep net and determine their identity. (Moths collected in pheromone traps are a poor indicator of which species of larvae will predominate in crops.) As a guide, catch at least 30 *Helicoverpa* spp. moths at random throughout the crop and on flowering weeds in the paddock (ignore the other moths such as tobacco loopers, brown cutworms and common armyworms that may fly in company with the native budworms). [26]

7.8.5 Preliminary threshold for *Helicoverpa* in canola

To date, thresholds available for managing *Helicoverpa* in canola have been based on 'best guesses'. In 2015 QDAF researchers conducted a replicated trial to determine the consumption rate of *Helicoverpa* larvae in canola. A consumption rate is a vital component of an economic threshold, and is an estimate of the yield and crop loss likely to occur. It is calculated from the lifetime consumption of a larva. In other words, it is a reflection of the amount of yield loss that would occur if the larva was not controlled. [27]

The trial involved confining individual larvae on canola racemes and allowing them to feed until they pupated. These data were compared with data from racemes that had no *Helicoverpa* feeding. In summary, results of the trial are:

i) The consumption rate of a *Helicoverpa* larva is estimated to be 2.4 grams of grain per larva.

ii) On average a larva damaged 10.5 pods and consumed 124 seeds.

26 K Hertel, K Roberts, P Bowden (2013) Insect and mite control in field crops. NSW DPI Management Guide. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0005/284576/Insect-and-mite-control-in-field-crops-2013.pdf

27 M Miles, A Quade, R Lloyd, P Grundy (2016) Recent insect pest management research findings and the application of results in the field. GRDC Update Papers 23 February 2016, https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/02/Recent-insect-pest-management-research-findings-and-the-application-of-results-in-the-field

iii) Larvae showed no preference for pod size/maturity.

Using the following equations, the potential yield loss and economic thresholds can be calculated for a range of crop and cost of control values – presented in Tables 5 and 6 below.

Potential yield loss (t/ha) per larva = D × P × V

Where

D = estimated yield loss per larva (t/ha)

P = pest density per sampling unit (e.g. per m^2)

V = crop value ($/t)

Economic threshold (larvae/m^2) = C / (V × D)

Where

C = cost of control ($/ha)

V = crop value ($/t)

D = estimated yield loss per larva (t/ha)

Table 5: *Potential yield loss ($/ha) from Helicoverpa larvae in canola.*

Crop value ($/t)	Number of larvae per m^2			
	1	2	3	4
400	9.6	19.2	28.8	38.4
450	10.8	21.6	32.4	43.2
500	12.0	24.0	36.0	48.0

Table 6: *Economic threshold (larvae per m^2) of Helicoverpa in canola.*

Cost of control ($/ha) (application + insecticide)	Crop value ($/t)		
	400	450	500
20	2.1	1.9	1.7
25	2.6	2.3	2.1
30	3.1	2.8	2.5
35	3.6	3.2	2.9
40	4.2	3.7	3.3

These thresholds are preliminary; the next stage is to repeat the trial as well as to evaluate the effectiveness of the proposed thresholds in adequately preventing economic yield loss in commercial crops. It is expected that there will be greater confidence and firm thresholds within 1–2 seasons. [28]

7.9 Other soil pests

There are several species of earwigs that may attack seedling crops including canola, cereals and legumes. The European earwig (*Forficula auricularia*) is the main culprit; an introduced species that appears to be spreading in southern agricultural areas. Their spread is likely to be at least partly due to increased levels of stubble retention. European earwigs chew developing seedlings and slow plant development. The typical appearance of damage is shredded leaf tips and/or irregular holes in leaves. They can also damage the pre-ripening grain heads of wheat. European earwigs

28 M Miles, A Quade, R Lloyd, P Grundy (2016) Recent insect pest management research findings and the application of results in the field. GRDC Update Papers 23 February 2016, https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/02/Recent-insect-pest-management-research-findings-and-the-application-of-results-in-the-field

range from 12–20 mm long, are smooth and shiny dark brown in colour with pale yellow legs.

It is important to distinguish earwig species in order to make the most appropriate management decision and accurately assess the risk of attack to emerging crop seedlings. For example, there is one native species, the common brown earwig (*Labidura truncata*), which is exclusively a beneficial insect that feeds on soft-bodied insects such as caterpillars, lucerne flea and mites. The common brown earwig can be distinguished by the presence of an orange coloured triangle behind the head on the elytra or 'wing-case'.

Control options for earwigs are limited but there is good evidence that insecticide seed treatments with fipronil will help crop seedlings withstand attack. Cracked grain baits (wheat, sorghum, corn) containing chlorpyrifos and sunflower/vegetable oil may also be used to control pest earwigs in some states. [29]

A number of soil dwelling insect pests such as cutworms, wireworms, bronzed field beetle, cockchafers and false wireworms have damaged emerging canola seedlings in recent years. In severe cases, plant stands can be thinned to such an extent that the paddock requires re-sowing. Occurrence of these pests is difficult to predict, so advice on their control should be sought before sowing if any problems are foreseen. The most severe damage tends to occur in crops following pasture, or where stubble has been retained. [30]

29 G McDonald, A Govender, P Umina (2016) Pest wrap up 2015 establishment pests, aphids, caterpillars and beneficials. GRDC Update Papers 16 February 2016, https://grdc.com.au/Research-and-Development/GRDC-Update-Papers/2016/02/Pest-wrap-up-2015-establishment-pests-aphids-caterpillars-and-beneficials

30 P Matthews, D McCaffery, L Jenkins (2016) Winter crop variety sowing guide 2016. NSW DPI Management Guide. NSW Department of Primary Industries, http://www.dpi.nsw.gov.au/__data/assets/pdf_file/0011/272945/winter-crop-variety-sowing-guide-2016.pdf