



CSIRO Entomology



## FINAL REPORT

### **CSE86C: Quantifying behavioural responses of *Helicoverpa* moths to trap crops for areawide management.**

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*A final report prepared for the Cotton Research and Development Corporation*

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**REPORTS**

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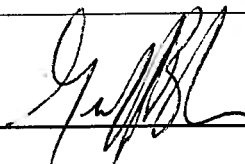
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## ***Part 3.3 – Final Report***

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### **1. Introduction and background**

Trap cropping is an established technology within many IPM systems for a broad range of crops. Successful trap crops act as diversionary hosts for key pests, attracting and concentrating the pests and their eggs into a relatively small area where the population can be controlled. Typically trap crops are 'suicide' crops that are destroyed prior to harvest in order to kill pests trapped within them. Trap cropping programs aim to reduce egg densities on the primary crop(s) on an areawide basis, resulting in an overall reduction in management costs for individual farms.

Interest in establishing areawide trap cropping programs has grown within the Australian cotton industry, following the success of the Central Queensland trap cropping program initiated by Dr Richard Sequeira as part of the INGARD® resistance management plan. Results suggested that substantial populations of *Helicoverpa* could be captured and destroyed by chickpea and pigeon pea trap crops.

The current project set out to quantify the effectiveness of trap crops and measure the impact of trap crops on egg densities in surrounding cotton. Other aims included quantifying the spatial and temporal nocturnal behaviour of moths in relation to trap crops and to cotton, and to evaluate the relative effectiveness of different spatial configurations of trap crops. The optimal dimensions for trap crop fields are unknown, and the proportion of the total cotton area that should be sown to trap crops is also uncertain. *Helicoverpa* moths have high mobility - their unassisted flight speed is about 5 metres per second, and with their tendency to fly downwind they may exceed 15 metres per second. At these velocities patches and strips of trap crops within bulk fields of cotton may be 'overshot' before individual moths have time to recognise them and respond. Furthermore, small patches of trap crop have proportionally more perimeter edge per unit area than large blocks. This may affect the 'holding power' of trap crops because moths within a trap crop will encounter and fly over the edge more frequently in small blocks than they would in larger blocks. These issues are also relevant to the design and deployment of INGARD® refuges.

This project also set out to address the integration of trap crop phenology with seasonal *Helicoverpa* population dynamics. For each of the main cotton growing valleys, the HEAPS model was used to predict the timing of *Helicoverpa* spring emergence from diapause, and also the autumn induction of diapause at the end of the season. The APSIM crop model was also used to predict the likely flowering times of chickpea planted over a range of sowing dates, based on historical meteorological data.

## 2. Project Objectives

1. To quantify the effectiveness of trap crops.

Achieved: Over the course of the project regular sampling was conducted in over 50 trap crops to collect data on the temporal incidence and abundance of *Helicoverpa* eggs, larvae and pupae. This data clearly shows the effectiveness of spring, mid-season and late summer trap crops.

2. To quantify the impact of trap crops on egg densities in surrounding cotton.

Achieved: At each trap crop site sampling was also undertaken in the associated cotton crops, so that *Helicoverpa* densities could be directly compared. At some sites transects were completed to quantify egg densities in the cotton at a range of distances away from the trap crop.

3. To quantify egg production and temporal behaviour of moths within trap crops and in associated cotton crops.

Achieved: Night observations were undertaken to quantify *Helicoverpa* moth activity levels in both trap crops and their associated cotton. Counts of egg densities at each site were completed the morning after each night observation. Light traps were used to quantify the hourly incidence and abundance of each species of *Helicoverpa* moths relative to other night flying insects, and to quantify the activity patterns of male and female moths.

4. To apply the HEAPS simulation model in order to evaluate and improve area wide management strategies based on trap cropping.

Achieved: Modelling studies were undertaken to predict the timing of *Helicoverpa* spring emergence from diapause and the autumn induction of diapause at the end of the season. With respect to spring chickpea trap crops this information was matched to predictions of crop phenology generated by the APSIM crop model based on historical meteorological data. The HEAPS model was also used to predict larval development times at various localities and times of the season.

## 3. Methods

### (a) Trap crops

Four main types of trap crops were examined over the course of the project: Chickpeas (cultivars: Amethyst, Barwon, Sona and mixed Kabuli types); Pigeon peas (Quest); Sorghum (Western red); and Sunflower (Advance). One crop of Mungbeans was also sampled. The chickpea trap crops were sown late in winter and timed to flower during spring in September and October at a time corresponding to the arrival of immigrating *H.punctigera* and the local emergence of *H.armigera* moths from over-wintering populations. This time also corresponds to the emergence of cotton seedlings. The pigeon peas, sunflower and mungbeans were sown in spring and timed to flower in mid-summer. The sorghum trap crops were sown in late spring to early summer and were timed to flower in late summer and early autumn, corresponding to the cut-out and defoliation of cotton crops. Observations and samples were also collected from Cotton crops associated with each trap crop.

(b) *Study sites*

During the three seasons spanned by the project, most observations and sampling were undertaken on 4 farms in the Namoi Valley: *Lowana*, *Auscott*, *Yarral* and the Australian Cotton Research Institute (*ACRI*). A number of trap crop fields on each farm were used in each season. In 1999 observations were also undertaken at *Myambra* near Collarenebri in the Gwydir valley. Table 1 provides a complete listing of the type, layout, dimensions, area, and time of season for all the trap crop fields from which observations and samples were collected on a regular basis. In the case of the Pigeon pea crops on *Yarral* in 1999-2000 and 2000-2001, they were grown as commercial crops for harvest rather than as trap crops per se. However because they were unsprayed for *Helicoverpa*, they were treated as being identical to trap crops for the purposes of this project.

(c) *Sampling Helicoverpa eggs and larvae*

Counts of *Helicoverpa* egg and larvae densities were made at 7 to 14 day intervals in each trap crop and in its associated cotton crop. Sampling was conducted visually on cotton and pigeonpea, and by both visual counts and sweepnet in chickpea. Visual sampling (Photos 1 and 2) involved carefully inspecting every part of whole plants in one metre sections of crop row to count the number of *Helicoverpa* eggs and larvae present. Five to six metres were sampled in each crop per check. Sweep net sampling (Photo 3) involved vigorously sweeping the net along and through the crop canopy whilst walking along the crop row. Each pair of forward and backward passes was counted as two sweeps, and each field check comprised a total of 100 sweeps, usually completed as 4 sets of 25 sweeps with the contents of the net examined after each set. Sorghum was sampled visually prior to head emergence. After head emergence, sorghum crops were sampled by collecting 30 random heads. The heads were processed in the laboratory by spinning each head into a bucket and examining the material shaken off under a magnifying glass to count eggs and larvae. To determine species (*H.armigera* or *H.punctigera*), incidence of parasitism and NPV infection status, the collected larvae were placed on artificial diet in the laboratory and reared at 25°C until they emerged as moths, or until a parasite emerged or they died.

At a number of sites, counts of *Helicoverpa* egg and larval densities were made in cotton crops at a range of distances away from the trap crop. Typically counts were made in the trap crop, and at 5, 50, 100 and 200 metres into the cotton away from the border with the trap crop.

(d) *Sampling Helicoverpa pupae*

Counts of *Helicoverpa* pupae densities in the soil beneath trap crops were made in trap crops in which large larvae were detected, in trap crops that had been slashed, and in cotton crops associated with trap crops. Typically 14 square metres were randomly sampled for pupae in each crop. A trowel was used to scrape the surface soil away from 1 square metre straddling the crop plant line (Photo 4). The subsurface was examined for the tell-tale emergence tunnels associated with pupating *Helicoverpa*. Tunnels were excavated to determine whether there was a live pupae present, or the remains of a pupal case. Emerged pupal cases were examined to determine whether a moth had emerged (by splitting the case longitudinally), or a parasitic wasp (by a circular decapitation of the pupal case). Once all tunnels had been excavated, the remaining soil in each metre being sampled was dug up to a depth of 5cm searching for any further pupae or pupal cases.



**Photo 1.** Visually checking a chickpea trap crop for *Helicoverpa* spp. Emerging cotton seedlings can be seen in the background in the adjacent cotton crop.



**Photo 2.** *Helicoverpa* egg on chickpea. The high number of small pinnate leaves with serrated edges make it very difficult to visually find eggs on chickpea plants.



Photo 3. Using a sweepnet to sample for *Helicoverpa* larvae in a chickpea trap crop



Photo 4. Using trowels to sample for *Helicoverpa* pupae under a chickpea trap crop

**Table 1.** A list of the major field sites used over the course of the project 1999 to 2002.

Season	Farm	Field	Trap crop	Period	Layout	Size (metres)	Area (Ha)
1999/2000	ACRI	A2	Pigeon pea (Quest)	Summer	6 patches	12 x 12	0.086
1999/2000	ACRI	A2	Pigeon pea (Quest)	Summer	2 strips	12 x 180	0.432
1999/2000	ACRI	F18	Sunflower (Advance)	Summer	2 strips	4 x 210	0.168
1999/2000	ACRI	Leitch 1	Pigeon pea (Quest)	Summer	block	54 x 500	2.7
1999/2000	Auscott	F6	Chickpea	Spring	block	400 x 430	17.3
1999/2000	Auscott	F9	Chickpea	Spring	block	300 x 550	16.4
1999/2000	Auscott	F10	Chickpea	Spring	block	300 x 550	16.1
1999/2000	Auscott	F31	Sorghum (West Red)	summer 1	block	64 x 800	5.12
1999/2000	Auscott	F31	Sorghum (West Red)	summer 2	block	64 x 800	5.12
1999/2000	Lowana	F4	Chickpea (Amethyst)	Spring	block	200 x 800	16
1999/2000	Lowana	F4	Chickpea (Barwon)	Spring	block	200 x 800	16
1999/2000	Lowana	F5	Pigeon pea (Quest)	Summer 1	block	200 x 800	16
1999/2000	Lowana	F5	Pigeon pea (Quest)	Summer 2	block	200 x 800	16
1999/2000	Yarral	F11	Pigeon pea (Quest)	Summer	block	800 x 630	50.5
2000/2001	ACRI	A1	Chickpea (Amethyst)	Spring	6 patches	12 x 12	0.086
2000/2001	ACRI	A1	Chickpea (Amethyst)	Spring	2 strips	12 x 180	0.432
2000/2001	ACRI	A1	Chickpea (Amethyst)	Spring	block	80 x 180	1.44
2000/2001	ACRI	A1	Pigeon pea (Quest)	Summer	6 patches	12 x 12	0.086
2000/2001	ACRI	A1	Pigeon pea (Quest)	Summer	2 strips	12 x 180	0.432
2000/2001	ACRI	A1	Pigeon pea (Quest)	Summer	block	80 x 180	1.44
2000/2001	ACRI	F17	Sunflower (Advance)	Summer	9 strips	4 x 210	0.756
2000/2001	ACRI	Leitch 2	Chickpea (Sona)	Spring	block	24 x 680	1.6
2000/2001	ACRI	Leitch 2	Chickpea (Kabuli)	Spring	block	24 x 680	1.6
2000/2001	ACRI	Leitch 2	Chickpea (Amethyst)	Spring	block	24 x 680	1.6
2000/2001	ACRI	Leitch 2	Pigeon pea (Quest)	Summer	block	72 x 680	4.8
2000/2001	Auscott	F8	Sorghum (West Red)	Spring	block	240 x 970	23.6
2000/2001	Auscott	F23	Soybean	Spring	block	190 x 190	36.9
2000/2001	Lowana	F1	Chickpea (Amethyst)	Spring	block	180 x 500	9
2000/2001	Lowana	F1	Chickpea (Sona)	Spring	block	180 x 500	9
2000/2001	Lowana	F1	Pigeon pea (Quest)	Summer 1	block	150 x 500	7.5
2000/2001	Lowana	F1	Pigeon pea (Quest)	Summer 2	block	150 x 500	7.5
2000/2001	Lowana	F1	Pigeon pea (Quest)	Summer 3	block	150 x 500	7.5
2000/2001	Lowana	F1	Pigeon pea (Quest)	Summer 4	block	150 x 500	7.5
2000/2001	Yarral	F9	Pigeon pea (Quest)	Summer	block	300 x 1190	35.7
2001/2002	ACRI	A2	Chickpea (Amethyst)	Spring	6 patches	12 x 12	0.086
2001/2002	ACRI	A2	Chickpea (Amethyst)	Spring	2 strips	12 x 180	0.432
2001/2002	ACRI	A2	Chickpea (Amethyst)	Spring	block	80 x 180	1.44
2001/2002	ACRI	A2	Pigeon pea (Quest)	Summer	6 patches	12 x 12	0.086
2001/2002	ACRI	A2	Pigeon pea (Quest)	Summer	2 strips	12 x 180	0.432
2001/2002	ACRI	A2	Pigeon pea (Quest)	Summer	block	80 x 180	1.44
2001/2002	ACRI	Leitch 1	Chickpea (Amethyst)	Spring	block	24 x 500	1.2
2001/2002	ACRI	Leitch 1	Chickpea (Sona)	Spring	block	24 x 500	1.2
2001/2002	ACRI	Leitch 1	Pigeon pea (Quest)	Summer	block	24 x 500	2.4
2001/2002	Lowana	F2	Chickpea (Sona)	Spring	block	144 x 583	8.4
2001/2002	Lowana	F2	Chickpea (Amethyst)	Spring	block	144 x 583	8.4
2001/2002	Lowana	F2	Pigeon pea (Quest)	Summer 1	block	144 x 562	8.1
2001/2002	Lowana	F2	Pigeon pea (Quest)	Summer 2	block	160 x 508	8.14
2001/2002	Lowana	F2	Pigeon pea (Quest)	Summer 3	block	144 x 583	8.4
2001/2002	Lowana	F2	Mung bean	Summer	block	352 x 580	20.4

(e) *Sampling emerging Helicoverpa moths*

In four fields 'emergence tents' were installed in chickpea trap crop fields to quantify the number of moths emerging from pupae in the soil subject to different cultivation treatments (Photo 5). The tents covered an area of soil 2m wide x 6 metres long (6 square metres). A collecting jar with a funnel set in the entrance was placed in the top-most section of the tent, and served to collect emerging *Helicoverpa* moths. The tents were installed in October or November following the maturation of the chickpea trap crop. They were checked every 7 to 10 days to monitor the number of *H.armigera* and *H.punctigera* moths emerging from the different cultivation treatments.

(f) *Comparing different trap crop layout configurations*

During each of the three cotton seasons encompassed by this project, the sites at ACRI (Fields A1 and A2) were used to compare the effectiveness of patches, strips and blocks of trap crop relative to cotton and relative to each other (Photos 6 to 12). Each season the experiments were conducted on spring chickpea trap crops, followed by summer pigeon pea trap crops. Each season the following layouts of trap crops were deployed: 6 patches, (12 x 12 metres), 2 strips (12 metres x 180 metres) and one block (80 x 180 metres). In each case these layouts were embedded within a background of unsprayed conventional cotton. Replicated counts of *Helicoverpa* egg and larvae density were made in each patch, strip and block at 7 to 14 day intervals, using the methods described in (c) above.

(g) *Night observations*

Night-time counts of *Helicoverpa* moth activity levels were conducted at irregular intervals in each trap crop and its associated cotton. Counts were made by observing the number of moths flying between goal posts per 5-minute interval using night vision goggles supplemented by infra-red illumination (Photos 13 and 14). Counts were made from the level of the crop canopy. Most night observation sessions were carried out between 8pm and 1am. On two occasions observations were undertaken at intervals throughout the night from dusk to the following dawn. The morning after each night-observation session, visual counts of egg densities were carried out in the cotton in the vicinity of the observed area. Daytime counts of moth densities were also made at some sites by 'flushing' moths from the canopy of the cotton or trap crop using a 1m beat stick and counting the number of moths flushed from cover per 100 metres or crop row.

During each night observation session portable light traps were used to quantify the proportion of *Helicoverpa* moths active relative to other similar sized moths and other night flying insects, and to determine the sex-ratio of *Helicoverpa* captured. The light traps used a 100W blacklight fluorescent tube and they were placed on the ground within the crop 200 metres away from the observation area. The light traps were vertically oriented and were shielded by a steel cone so as to only intercept insects flying over them, and not to attract insects from further a field along a horizontal plane.

(h) *The timing of oviposition*

A field and laboratory experiment was undertaken to determine the times of night over which female *Helicoverpa* moths lay their eggs. In the field experiment, individual plants within a pigeon pea trap crop were covered by mesh bags to exclude access by egg-laying moths (Photo 15). At set intervals through the night, sets of plants were temporarily uncovered to expose them to egg laying moths for 1 hour periods.

In the laboratory experiment, 12 pairs of male and female moths were housed in plastic containers containing paper towel and honey solution, and placed in a controlled temperature cabinet. The light/dark regime was set to 14 L:10 D, and was reversed so that the moths experienced darkness during normal working hours. Each pair was given 3 days to acclimatise to the conditions and to allow the exclusion of any infertile pairs. Once each pair of moths had completed the acclimitisation period and were known to produce fertile eggs, counts of the number of eggs laid were made at set intervals after the onset of darkness inside the controlled temperature cabinet.

(i) *The distribution of Helicoverpa within a block of trap crop*

In a one-off experiment the spatial distribution of *Helicoverpa* larvae within a 5 Hectare block of pigeon pea trap crop was quantified by counting the number of larvae present per metre of row at 480 sites spread uniformly throughout the crop. Sampling sites were marked



**Photo 5.** Emergence tents were used to capture *Helicoverpa* moths emerging from pupae in the soil under a previous chickpea crop.



**Photo 6.** Patches (12 rows x 12 metres) of chickpea trap crop set within a cotton field at ACRI. The flowering chickpea provides an attractive host in contrast to the emerging cotton.



**Photo 7.** A strip (12 rows x 180 metres) of chickpea trap crop set within a cotton field at ACRI.



**Photo 8.** A block of chickpea trap crop set within a cotton field at ACRI. This trap crop has matured and is no longer attractive to *Helicoverpa*.



**Photo 9.** A patch (12 rows x 12 m) of pigeon pea trap crop set within a cotton field at ACRI



**Photo 10.** The same patch of pigeon pea trap crop shown in Photo 9. Both the cotton and pigeon pea are flowering and attractive to *Helicoverpa* moths as oviposition hosts.



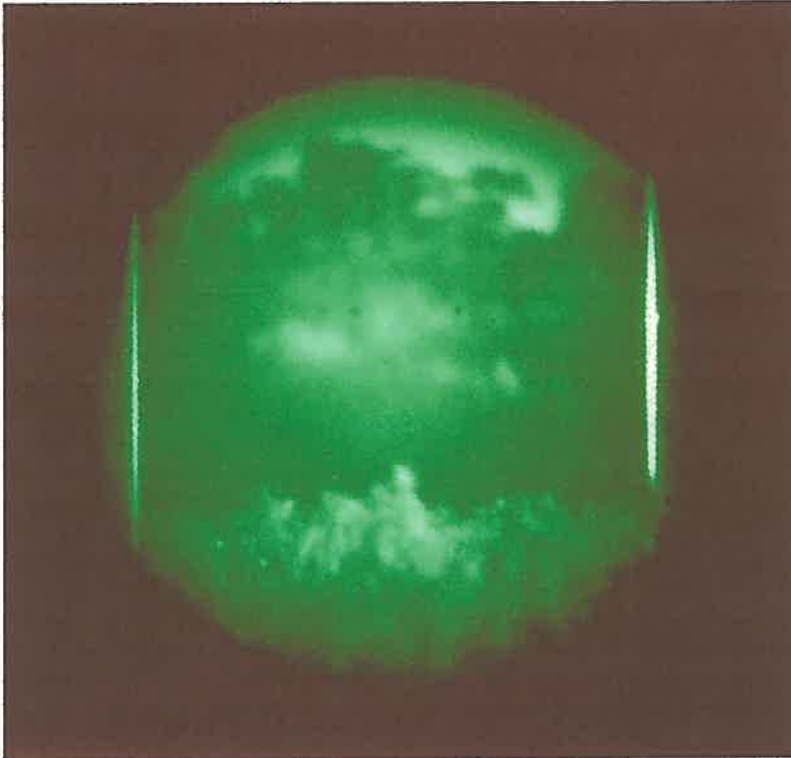
**Photo 11.** A flowering pigeon pea trap crop adjacent to a flowering cotton crop



**Photo 12.** A strip of sunflowers at ACRI. Despite the difference in height between the cotton and sunflower crops, the same *Helicoverpa* egg densities were counted on each crop.



**Photo 13.** Goal posts set up in a strip of pigeon pea at ACRI.



**Photo 14.** View of the same goal posts in Photo 13 through night vision goggles using supplementary infra red illumination.



**Photo 15.** Pigeon pea plants covered by egg exclusion bags. Individual plants were exposed to egg laying moths for a range of 1 hour intervals during the night.



**Photo 16.** Pigeon pea crops are most attractive to *Helicoverpa* during the period two days before the flower buds open. Note the eggs laid amongst the flower buds in the close-up on the right.

out with a measuring tape, in rows 10 metres apart and at 10 metre intervals along the length of each row. 10 observers used beat sheets to sample 1 metre of pigeon pea at each sampling point. All *Helicoverpa* larvae were collected and placed into 70% ethanol for later analysis at the University of Queensland to detect DNA microsatellite markers. Prior to collecting the samples, a comparison was made between visual counting, beat sheets and a sweep net to determine the most efficient method. The beat sheet method was adopted because it detected substantially more *Helicoverpa* larvae in pigeon pea, and was also faster than the other two methods.

(j) *The timing of Helicoverpa pupal diapause relative to trap crop phenology*

The HEAPS model was used to predict the timing of spring emergence of *H.armigera* moths from their over-wintering diapause. Graphs and tables of emergence times were used to formulate recommendations for the planting and flowering times of spring trap crops. The HEAPS model was also used to predict the timing of autumn diapause induction, relative to the time of flowering of late-summer trap crops.

The APSIM model was used to predict the probable flowering times of chickpea trap crops at a range of locations and under full and half moisture profiles.

(k) *A modelling evaluation of the total area of trap crops required for effective operation.*

A spreadsheet model was devised to compute the likely proportion of *Helicoverpa* eggs within a discrete region expected to be laid onto trap crops. The model was used to generate the predicted proportions of total eggs that will be laid onto trap crops for 5 different total areas of trap crops: 1%, 2%, 5%, 10% and 20% of the total crop area. The model generates predictions for a range of levels of trap crop attractiveness relative to cotton. Specifically, the levels of trap crop attractiveness considered were within the ranges 1 to 10, and 10 to 100 times as attractive as cotton. For example, a value of 1 indicates the trap crop is identical in attractiveness to cotton, and that it would receive the same number of eggs per unit area. A value of 10 indicates that female moths find the trap crop 10 times more attractive than cotton, and therefore would tend to lay 10 times as many eggs onto the trap crop. However the model assumes that the moths disperse homogeneously throughout the area, and that the incidence of encountering the trap crop is directly proportional to the total area of trap crop within the region. The model assumes that only trap crops and cotton provide hosts for *Helicoverpa* in the region. The scale of the model's predictions are undefined.

The equations used by the model are as follows:

$$E = N/(N+1)*100$$

Where...

E = % eggs laid on trap crops

N = Number of eggs laid into a trap crop per 1 egg in cotton

A = Attractiveness of the trap crop relative to cotton

T = Total area of Trap crop within the region

And where N is computed as follows:

$$N = (A*(T/100))/(1*((100-T)/100))$$

#### 4. Results

##### *The effectiveness of trap crops.*

All the trap crops examined in the course of the project successfully attracted *Helicoverpa* eggs, and the results for each type of crop were generally consistent across the different field sites and across the three cotton seasons 1999/00, 2000/01 and 2001/02. The trap crops were most attractive whilst they were flowering, or, in the case of pigeon pea, in the 2 days immediately prior to flowering. The different types and seasonal timing of trap crops varied in their effectiveness at diverting eggs away from cotton.

##### *(i) Spring flowering trap crops*

Spring flowering chickpea proved to be extremely attractive to *Helicoverpa*, with densities of up to 25 eggs and/or larvae per row metre commonly occurring. Figure 1 illustrates a typical example of *Helicoverpa* densities in a chickpea trap crop monitored over a range of sampling dates. Chickpea trap crops attracted eggs of both *H.armigera* and *H.punctigera*, although because the flowering period corresponds with the influx of immigrating *H.punctigera* at the start of each season, this species tended to predominate in chickpea trap crops. In all cases the density of *Helicoverpa* was substantially higher in chickpea trap crops than densities on nearby newly emerged cotton crops (Figure 1).

During October and November spring trap crops of chickpea are usually the most attractive crop host for *Helicoverpa* available within the landscape, and they are likely to make a significant impact by diverting eggs of the first generation of *H.punctigera* and *H.armigera* away from cotton. By acting as a population sink early in the season, and suppressing population size, spring trap crops are likely to make an impact on the subsequent dynamics of these pests for the next generation and possibly longer. By attracting the eggs of *H.armigera* moths that have emerged from over-wintering diapause, spring chickpea trap crops are also likely to play an important role in reducing the incidence and severity of resistance to insecticides and transgenic cotton. Moths emerging from diapause are the survivors of the last generation at the end of the previous cotton season. Individuals from this generation are likely to be resistant to many insecticides, and in fact this population of emerging moths are the sole carriers of resistance from one season to the next.

Spring chickpea trap crops required careful monitoring to ensure that they did not become nurseries generating *Helicoverpa* moths. Apart from occasional NPV infection or parasitism by Tachinid flies, *Helicoverpa* larvae generally had very high survival rates to the pupal stage in chickpea trap crops. Appendix 5: "Spring trap crop management guidelines" addresses this and other management issues.

##### *(ii) Mid-summer trap crops*

The *Helicoverpa* population data collected in this project consistently showed a high degree of variation in the apparent attractiveness of summer flowering trap crops relative to the egg densities recorded in nearby cotton. For example, although the 'Quest' pigeon pea cultivar is indeterminate and continues producing flowers throughout the season, it does not maintain a constant level of attractiveness. In most of the pigeon pea trap crops examined in the course of this project, the highest densities of white eggs were recorded during the 2 days immediately prior to the bulk of the field commencing flowering (Photo 16). Figure 2 shows the rises and falls in egg densities in three staggered plantings of pigeon pea at Lowana. Block 1 was planted first, then block 2 was planted 3 weeks later, and block 3 was planted 3 weeks after block 2. While the cotton maintained relatively constant egg densities (see Photo 17). As each planting of pigeon pea became highly attractive, there was also an increase in

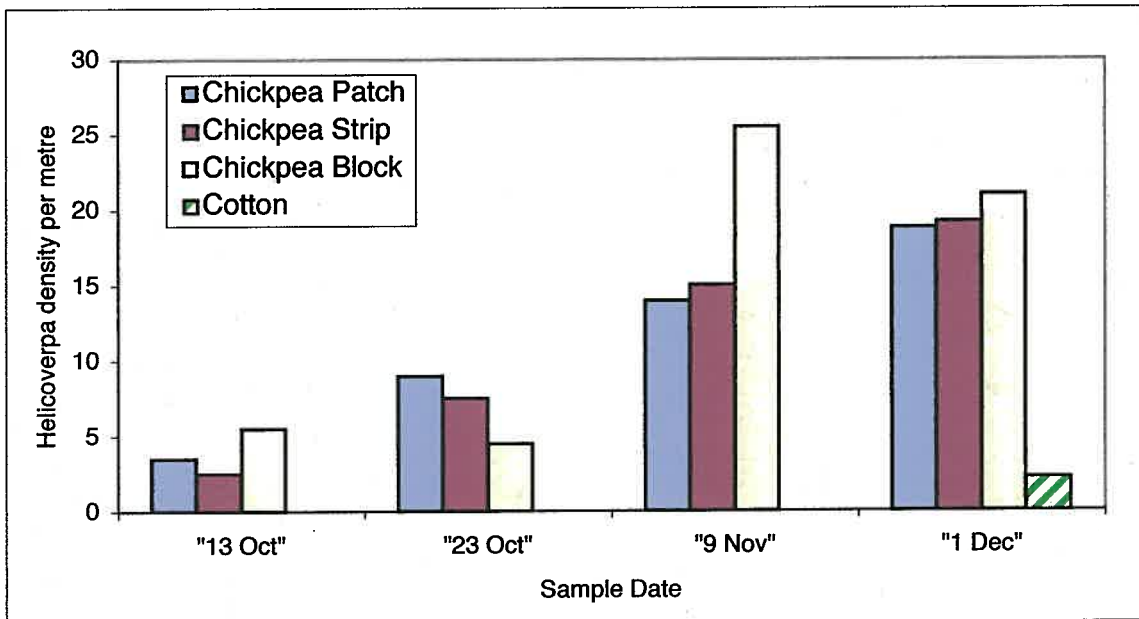


Figure 1. The density of *Helicoverpa* in Chickpea and Cotton at ACRI Field A1 over a range of sampling dates in the 2000/01 season. Cotton had not yet emerged from the ground until after the end of October.

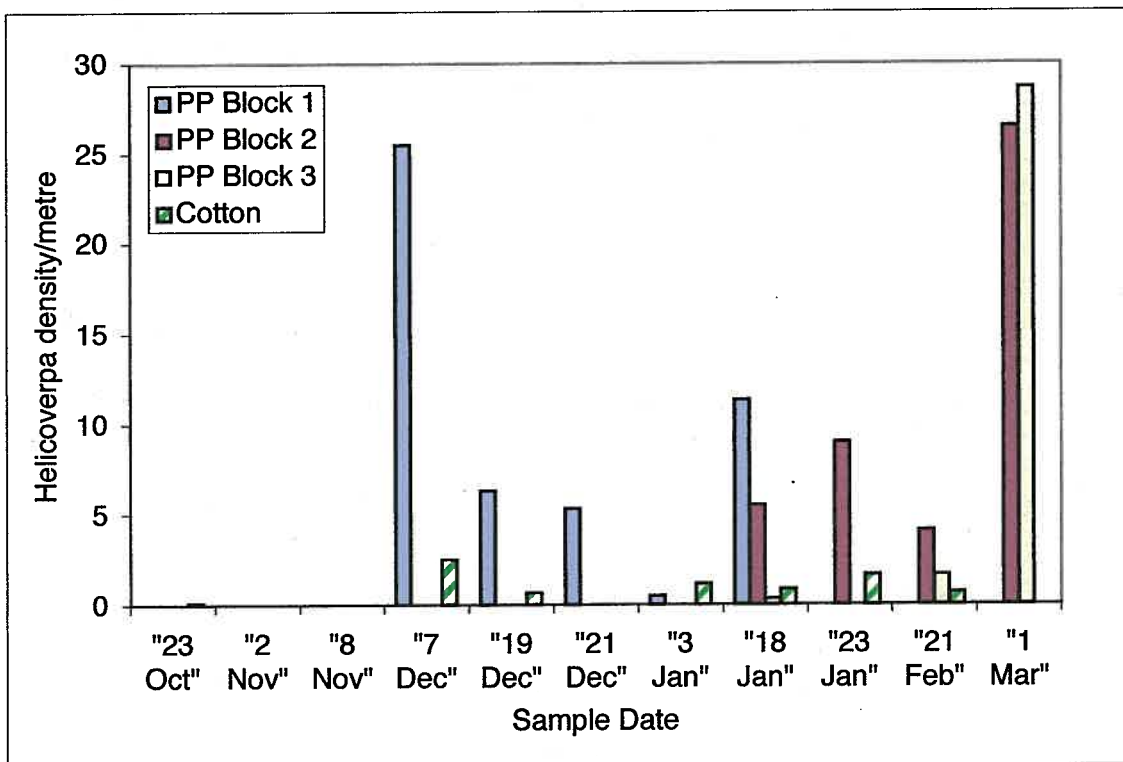


Figure 2. The density of *Helicoverpa* in Pigeon pea and Cotton at Lowana over a range of sampling dates in the 2000/01 season. The blocks were planted in sequence at approximately 3 week intervals.



**Photo 17.** *Helicoverpa* eggs and a larvae on a mature pigeon pea pod.



**Photo 18.** A *Helicoverpa* larvae that has died of NPV infection within a pigeon pea trap crop.

egg densities in the other adjacent blocks of pigeon pea as well, even though they had become less attractive and had previously shown declining egg densities. In contrast, Figure 3 presents data from a single block of pigeon pea trap crop at *Myambala* that consistently had a lower proportion of plants infested by *Helicoverpa* than the adjacent cotton. Eggs from both species of *Helicoverpa* were detected in pigeon pea trap crops at all sites.

The strips of sunflower trap crops at ACRI consistently had similar egg densities to the adjacent cotton. This is despite the substantial difference in height between the crop canopies (see Photo 12). Crop canopy height has been identified as an important component of host attractiveness by other research (Dr R.Sequeira pers. com). The single crop of mungbean at Lowana monitored during 2000/01 failed to attract *Helicoverpa* eggs on most sampling dates, and always hosted less *Helicoverpa* larvae than nearby cotton. However from M.Dillon's personal experience mungbeans at other locations in previous seasons have been known to support extremely high populations of up to 70 larvae per square metre. Eggs from both species of *Helicoverpa* were collected from the sunflower and mungbean trap crops.

NPV infection rates were relatively high in pigeon pea crops (see Photo 18). Predatory insects and spiders, and parasitic wasps were also common in the unsprayed pigeon pea and sunflower trap crops.

### (iii) *Late-summer trap crops*

Sorghum trap crops timed to flower at the end of the cropping season as the cotton crops were cutting out consistently attracted very high *H.armigera* egg densities, and they sometimes hosted extreme populations of *H.armigera* larvae (Photos 19 to 22). *H.punctigera* were never recorded from sorghum trap crops. Figure 4 presents data from a late season trap crop composed of two separate sowings of sorghum. Block 1 was planted 2 weeks before block 2. The maximum density of *Helicoverpa* eggs and larvae recorded was 256 per metre on 4 April 2001 in block 2. These trap crops were slashed when the larvae within each of them reached medium size, and the stubble was subsequently cultivated. Sampling in the soil below these crops did not detect any pupae.

Such late summer trap crops act as a population sink at the end of the season. As such they are likely to play a valuable role in providing a host for last-generation *H.armigera* to lay their eggs on – eggs that may otherwise be placed elsewhere in the landscape and eventually produce over-wintering diapausing pupae.

### *The effectiveness of cultivation to suppress moth emergence.*

The results from the 'emergence tent' trials are presented in Table 2. Areas of soil in 4 different chickpea trap crop fields that were not cultivated after the crop had finished produced up to 0.42 *Helicoverpa* moths per square metre, equivalent to over 4166 moths per hectare. In the portions of fields that were cultivated once with a disc plough, the density per metre was reduced at least 10-fold to between 0.01 and 0.04 moths, or between 139 and 417 moths per hectare. A double cultivation was evaluated in one chickpea field. The 36 square metres within the emergence tents that had been cultivated twice with a disc plough at different angles did not produce any live moths at all. Overall, the data clearly show that cultivating the soil once trap crops are no longer attractive is a very effective method of killing any surviving pupae, and that ideally two passes should be made to increase the efficacy of the cultivation.

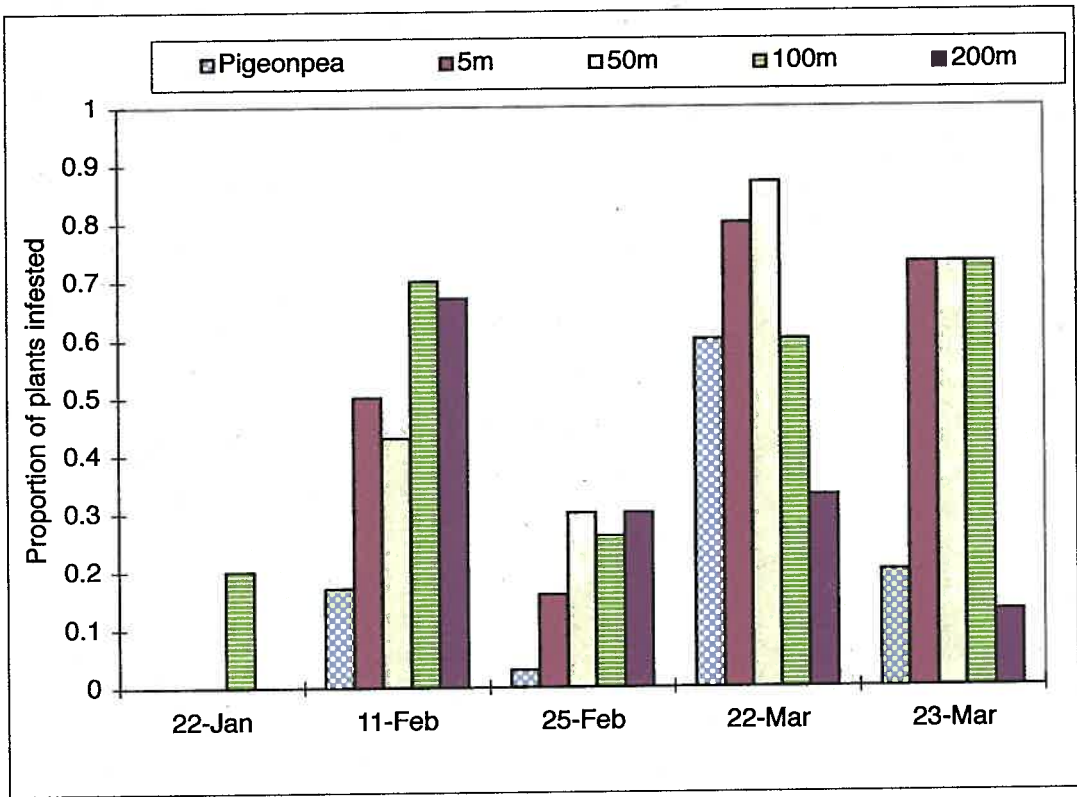


Figure 3. Myambra proportion of plants infested with *Helicoverpa* spp. white eggs: Pigeonpea, Cotton 5m, 50m, 100m, 200m

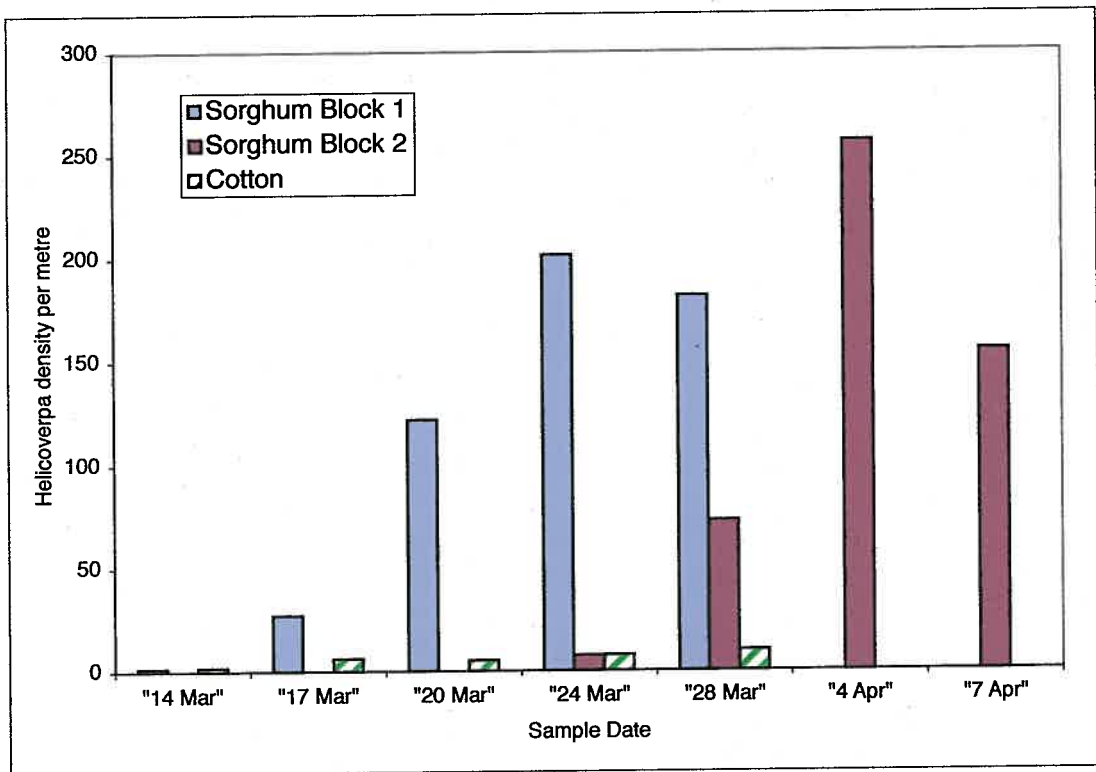


Figure 4. The density of *Helicoverpa* in Sorghum and Cotton at Auscott over a range of sampling dates in 2000/01 season in two staggered sowings of Sorghum sown 14 days apart.



**Photo 19.** A late season sorghum trap crop with heads emerging from their boots, adjacent to a mature cotton crop that has cut out.



**Photo 20.** Sorghum is highly attractive to *H. armigera* during the period as the head emerges from the boot (left) and the anthers fill (right). Once the head turns orange (centre) few eggs are laid on it. Most heads within a sorghum crop open within 7 days of each other, which results in a crop-wide window of high attractiveness about 12 days long.



**Photo 21.** A slashed portion of a staggered planting of sorghum trap crop at Auscott.



**Photo 22.** At least 7 *H. armigera* medium larvae can be seen crawling on the remains of a slashed late season sorghum trap crop. The field was subsequently cultivated, and pupae sampling did not detect any successful pupae in the soil.



**Photo 23.** Cultivating a chickpea trap crop to destroy *Helicoverpa* pupae.



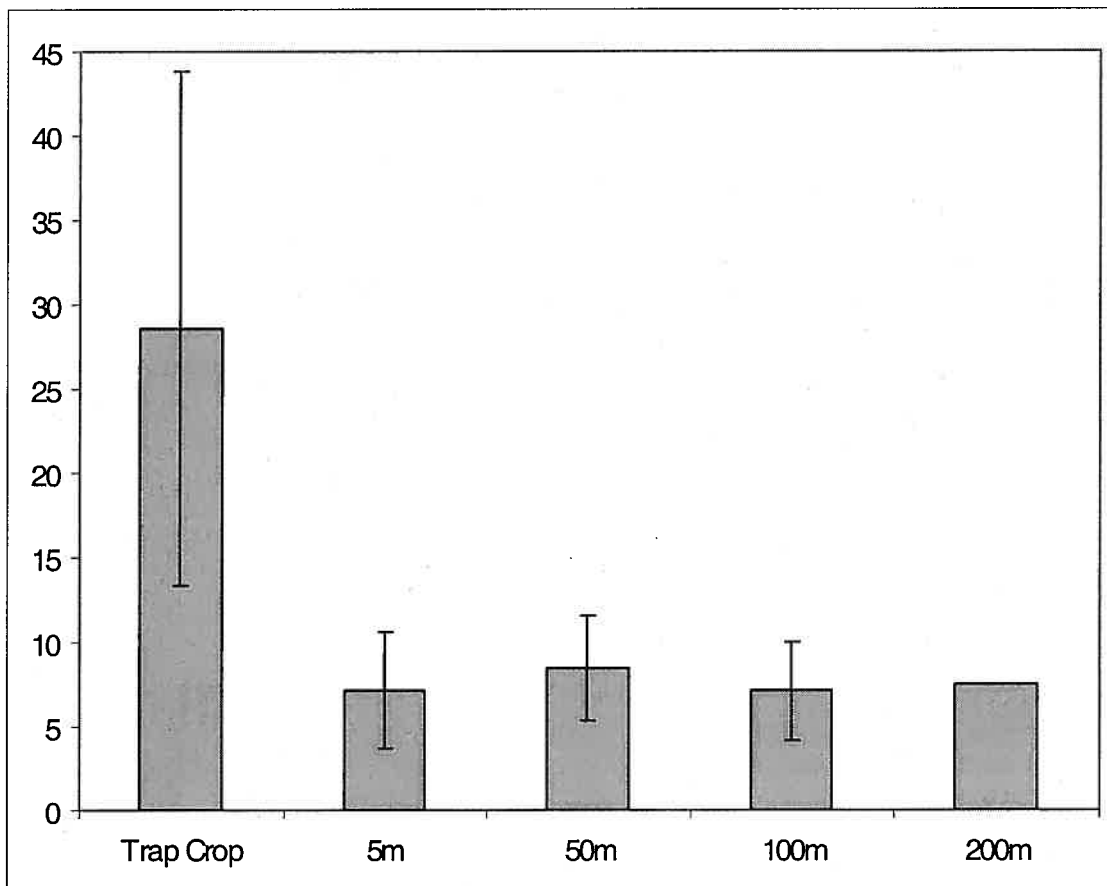
**Photo 24.** Cultivating a pigeon pea trap crop to destroy *Helicoverpa* pupae.

**Table 2.** The number of moths caught in emergence tents placed over different cultivation treatments following chickpea trap crops.

Auscott F6, F9, and F10	Uncultivated	1 x Cultivation	
No. of tents	6	12	
Square metres	36	72	
Total <i>Helicoverpa</i> moths	15	1	
Moth density/m	0.42	0.01	
Moths/Ha	4167	139	

Lowana Field 4	Uncultivated	1 x Cultivation	2 x Cultivation
No. of tents	6	4	6
Square metres	36	24	36
Total <i>Helicoverpa</i> moths	15	1	0
Moth density/m	0.42	0.04	0.00
Moths/Ha	4167	417	0



**Figure 5.** ACRI Pigeonpea egg density per metre in unsprayed cotton at a range of distances away from the trap crop.

### *The impact of trap crops on egg densities in surrounding cotton.*

On five occasions during the project replicated samples were taken at a range of distances away from trap crops into the surrounding cotton. Overall there was no evidence to suggest that there was any type of gradient in egg density as the distance away from a trap crop increases. Figure 5 presents data summarised over a whole season for *Helicoverpa* egg densities in a pigeon pea trap crop and in surrounding cotton at distances of 5, 50, 100 and 200 metres away from the trap crop. In this case the pigeon pea attracted significantly more *Helicoverpa* eggs than the cotton, however there are no differences in the density of eggs in the cotton relative to the distance away from the trap crop. Figure 3 presents data from *Myambra* in which the pigeon pea trap crop generally has a lower proportion of plants infested by *Helicoverpa* than the surrounding cotton at 5, 50, 100 and 200 metres away on 5 separate dates. The sampling dates span the period in which the pigeon pea trap crop commenced flowering and should have become an attractive host. The data show no consistent trend of either increasing or decreasing proportions of plants infested with *Helicoverpa* in relation to the distance from the trap crop.

### *The relative effectiveness of patch, strip and block trap crops.*

Our results consistently showed that solid blocks of trap crops are much more effective at attracting *Helicoverpa* eggs than strips or patches. Furthermore as the area of blocks of trap crop increased, they tended to host relatively higher densities of *Helicoverpa*. Figures 6 and 7 are presented as typical examples of the differences in *Helicoverpa* densities that were found in the different layouts. Figure 6 presents data from pigeon pea trap crops at ACRI. The patch and strip configuration attracted few eggs – and significantly lower densities than the surrounding cotton. However the block of pigeon pea attracted significantly more eggs than the nearby cotton. Figure 7 presents data on the density of *H.armigera* pupae under cotton, and under patches, strips and a block of an adjacent sorghum trap crop at ACRI. The block of sorghum hosted significantly more pupae than the strips or patches. However the cotton crop also had a similar density of pupae to the sorghum block.

There are a number of possible reasons that may contribute to this result. For example, because *Helicoverpa* moths fly at approximately 5 metres per second, it is likely that they may over-shoot narrow strips or small patches of suitable hosts – particularly if they are flying down-wind with wind assistance. Fast flying moths may either pass over without detecting the trap crop, or even if they do detect it, they may not be able to easily find it again in an area dominated by cotton. Another feature of narrow strips or small patches of trap crop is that they have a relatively high perimeter to area ratio. As a trap crop increases in size, the length of perimeter per unit of internal area declines. This means that for moths that are randomly flying within a trap crop, the probability of encountering the boundary and leaving the trap crop increases as the crop reduces in size in one or more dimensions. A long thin strip of trap crop will have a greater length or perimeter than a more square shaped trap crop of the same area.

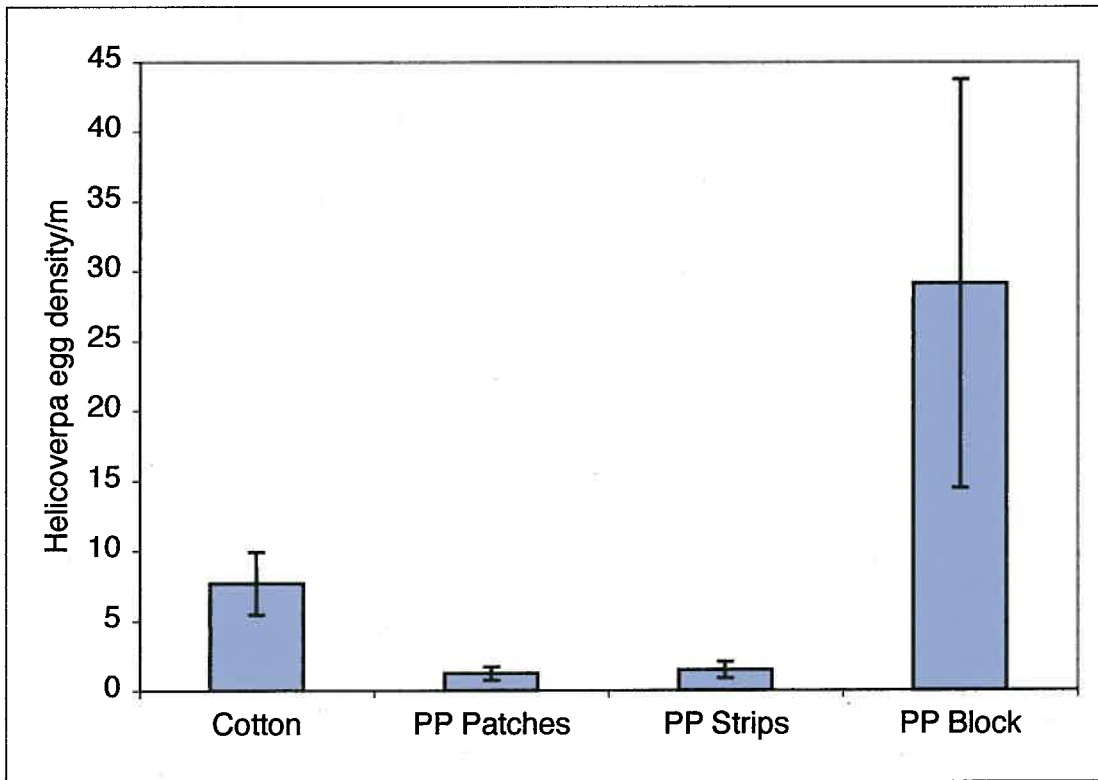


Figure 6. *Helicoverpa* egg densities in Pigeonpea patches, strips and block versus cotton at ACRI.

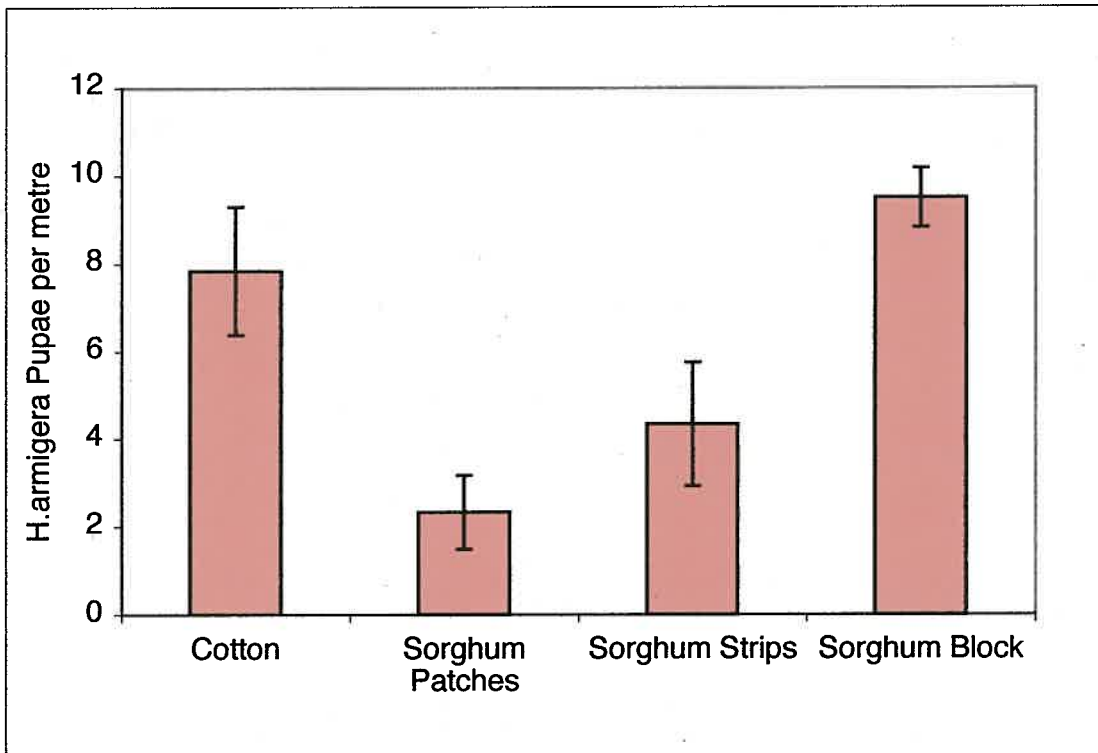


Figure 7. *Helicoverpa* pupae densities in Sorghum patches, strips and block versus cotton at ACRI.

### *Nocturnal observations of Helicoverpa moth activity.*

Results from the nocturnal observations indicate that *Helicoverpa* moths fly freely over trap crops and cotton and do not appear to restrict their movements to specific types of hosts. Figures 8 and 9 present typical data of *Helicoverpa* moth activity levels as measured by the number of moths passing through goal posts set within the trap crop and also within the adjacent cotton. The observations presented in Figure 8 show moth counts in the trap crop and in the adjacent cotton at 4 different distances from the trap crop. Figure 9 presents observations in the cotton, and at also 3 different distances within the adjacent trap crop. There is no consistent pattern of moth activity in relation to the type of crop or distance from the trap crop. Figure 8 also illustrates the range of moth activity levels recorded during the project, from zero to over 25 moths per minute flying through the goal posts. A consistent observation was that moth activity levels were highest at dusk and immediately after sunset. Figures 9a, 9b, 9c and 9d present moth activity levels in a pigeon pea trap crop and adjacent cotton at Yarral in a sequence over the course of an entire night. The data clearly show high activity levels early in the evening that steadily decline during the night through to dawn. Figure 10 summarises the whole night counts presented in figure 9, and shows the overall average moth activity level at each observation station. There are no significant differences between the cotton site and the three pigeon pea sites.

There were no consistent correlations between nocturnal *Helicoverpa* moth activity levels and *Helicoverpa* egg densities counted the following morning over the three seasons spanned by the project. A possible explanation for this result may be that moths are engaging in a range of behaviours apart from oviposition, including dispersing, feeding, searching for mates or seeking shelter. There were also no consistent correlations between nocturnal *Helicoverpa* moth activity levels over trap crops or cotton and the subsequent daytime flush counts of *Helicoverpa* moths sheltering in these crops the following morning. This is illustrated by comparing the flush counts shown in Figure 11 with the nocturnal moth activity counts for the previous nights presented in Figure 8 in a pigeon pea trap crop at Myambala, and in the adjacent cotton at 5, 50, 100, and 200 metres from the trap crop over 4 sampling dates.

Light trap catches indicated that there were distinct patterns of nocturnal activity by night flying insects in general, and by female and male *Helicoverpa* moths in particular. Figure 12 shows the number of night flying insects relative to *Helicoverpa* moths captured by a light trap that was checked at hourly intervals. The other night flying insects captured were predominantly crickets, beetles and other types of moths. Similar to *Helicoverpa*, other insects were most active around dusk and in the period following sunset. Figure 13 shows the relative proportion of *Helicoverpa* moths to these other insects during each time period. Figures 12 and 13 show that from around midnight onwards *Helicoverpa* moths became the most abundant night flying insects captured by the light trap, and the proportion of *Helicoverpa* moths relative to other insects continued to increase over the second half of the night.

Light trap data clearly show a marked difference in activity levels between male and female *Helicoverpa* moths. Figure 14 shows the rate of moths captured during each time interval through the night. Female moth activity levels are initially high but fall to a low level from before midnight through to dawn. In contrast male moth activity levels remain relatively constant throughout the night. Figure 15 shows the relative proportion of each sex. It shows a strong contrast as the night progresses, with proportionally more female moths initially, but then steadily decline relative to the proportion of males through the remainder of the night.

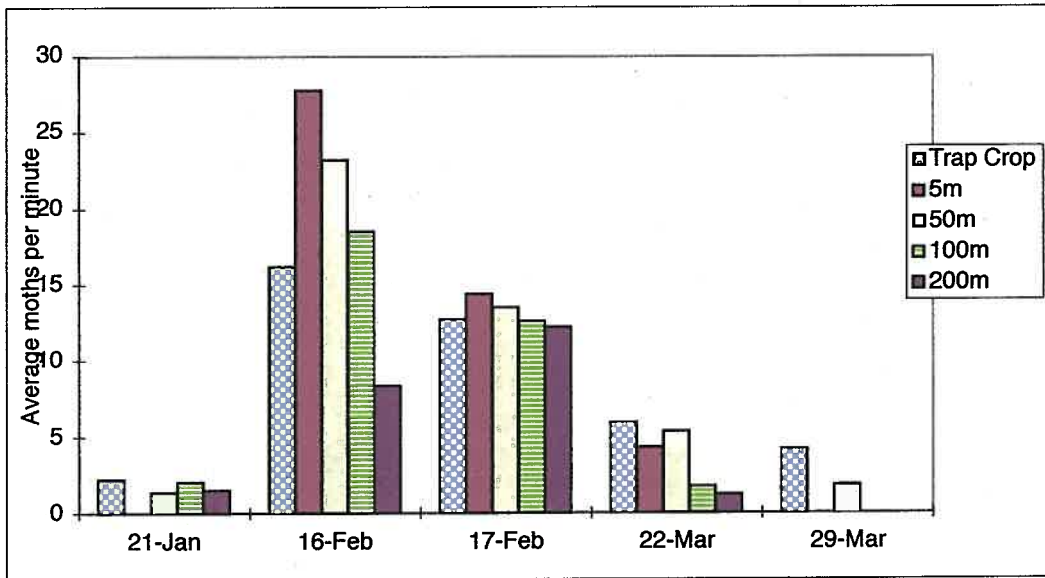


Figure 8. Myambra 1999 Night goal-post counts of moth activity in trap crop and in the cotton at 5m, 50m, 100m and 200m.

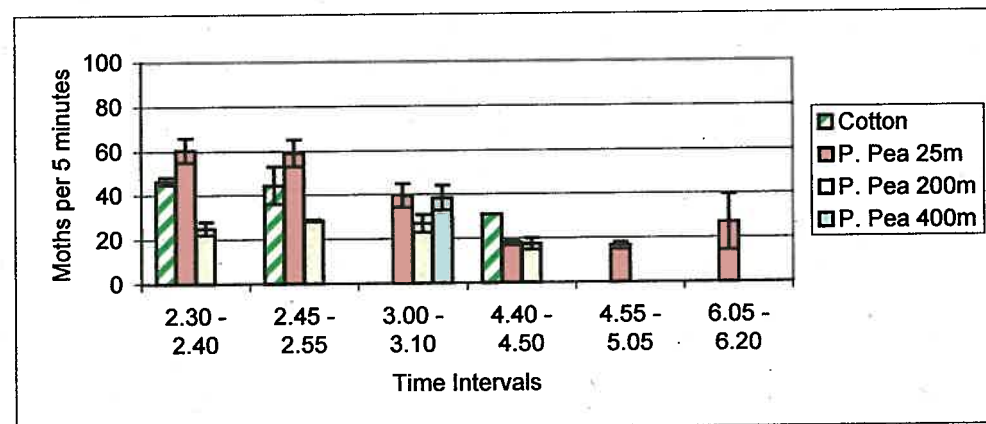
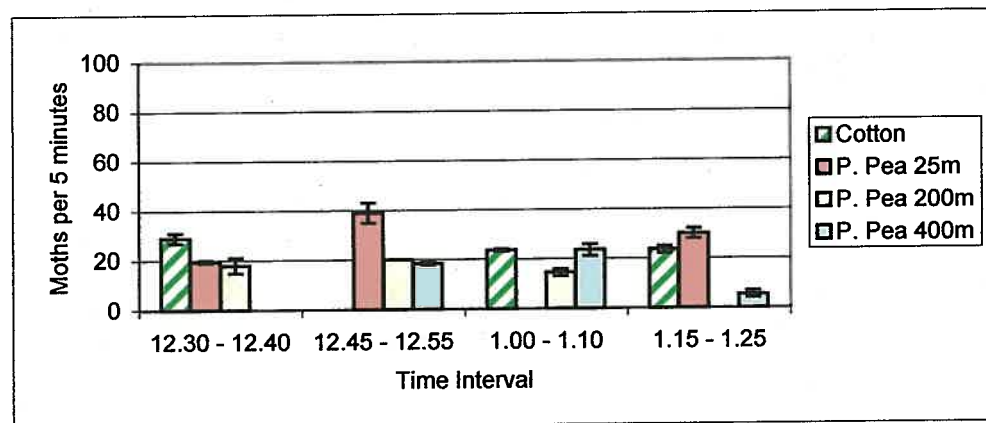
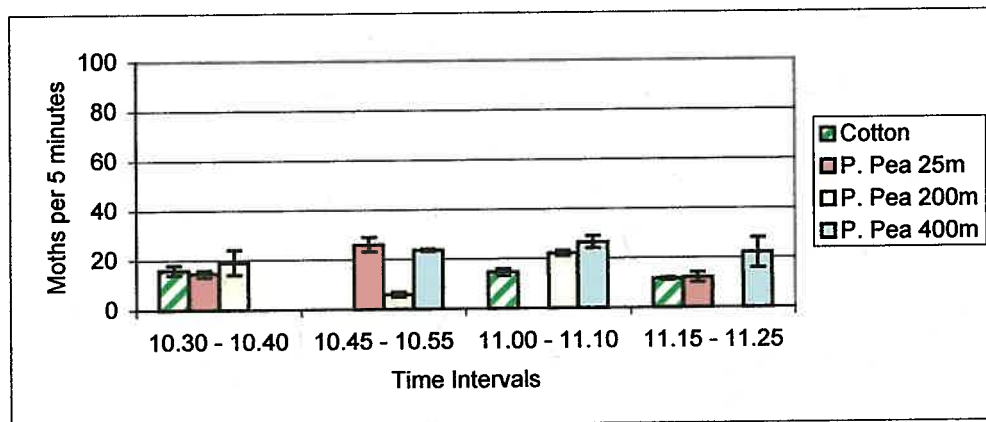
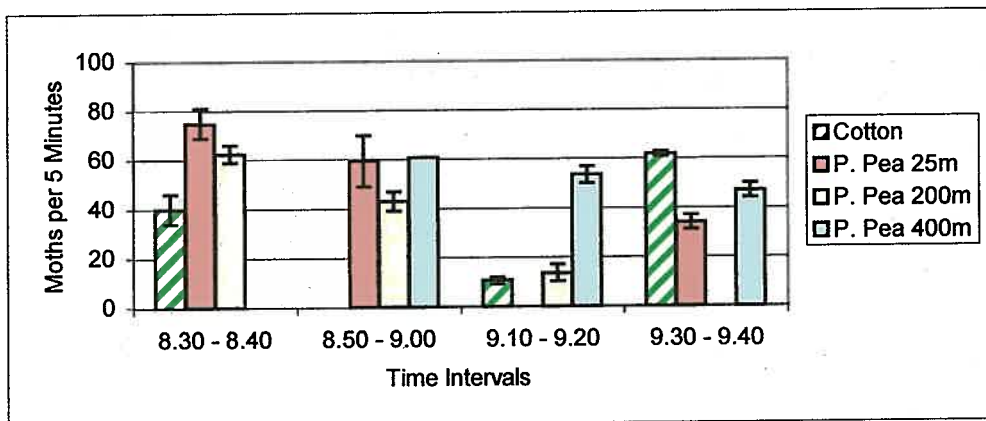


Figure 9. Yarral Night Observation 22-23/2/00 - showing intervals throughout the night.

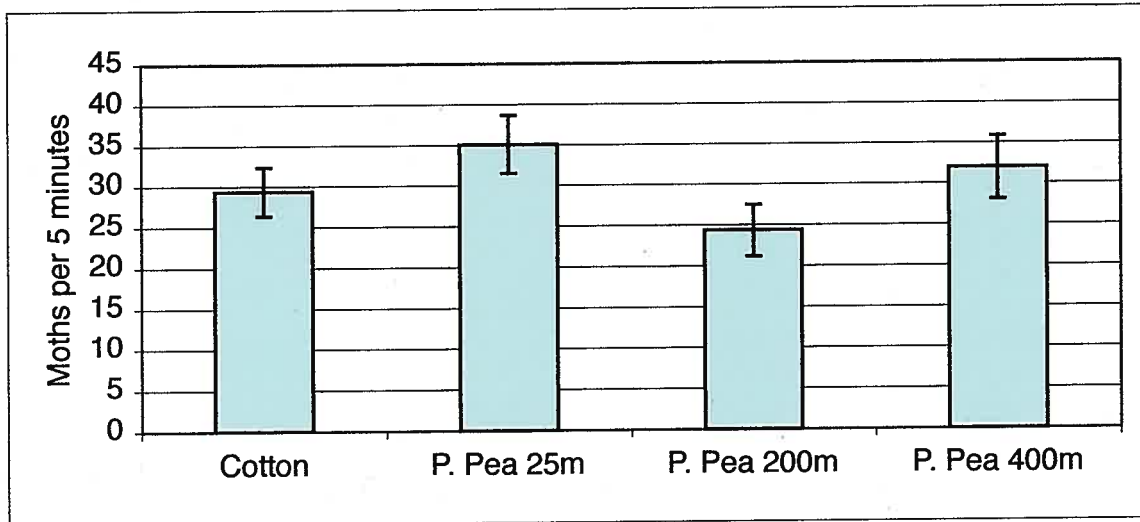


Figure 10. Yarral Night Observation 22-23/2/00 - Average moths per 5 minutes

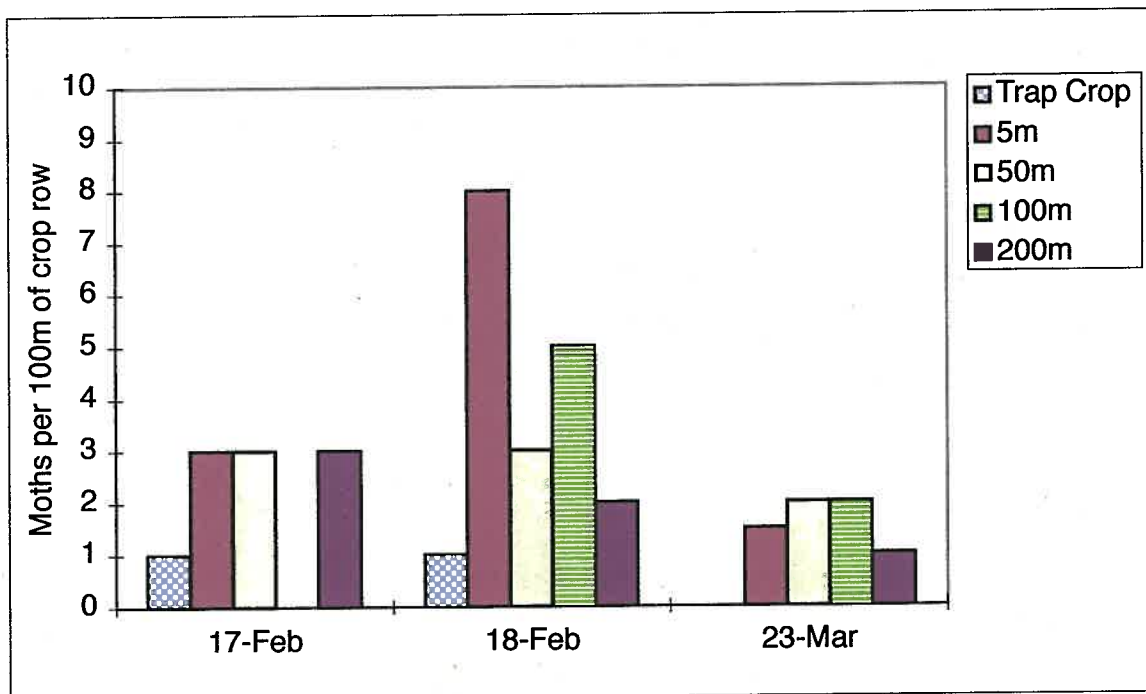


Figure 11. Daytime Flush Counts Myamblya Pigeonpea 1999 showing the number of moths counted in 100 metre flush transects at range of distances away from the trap crop.

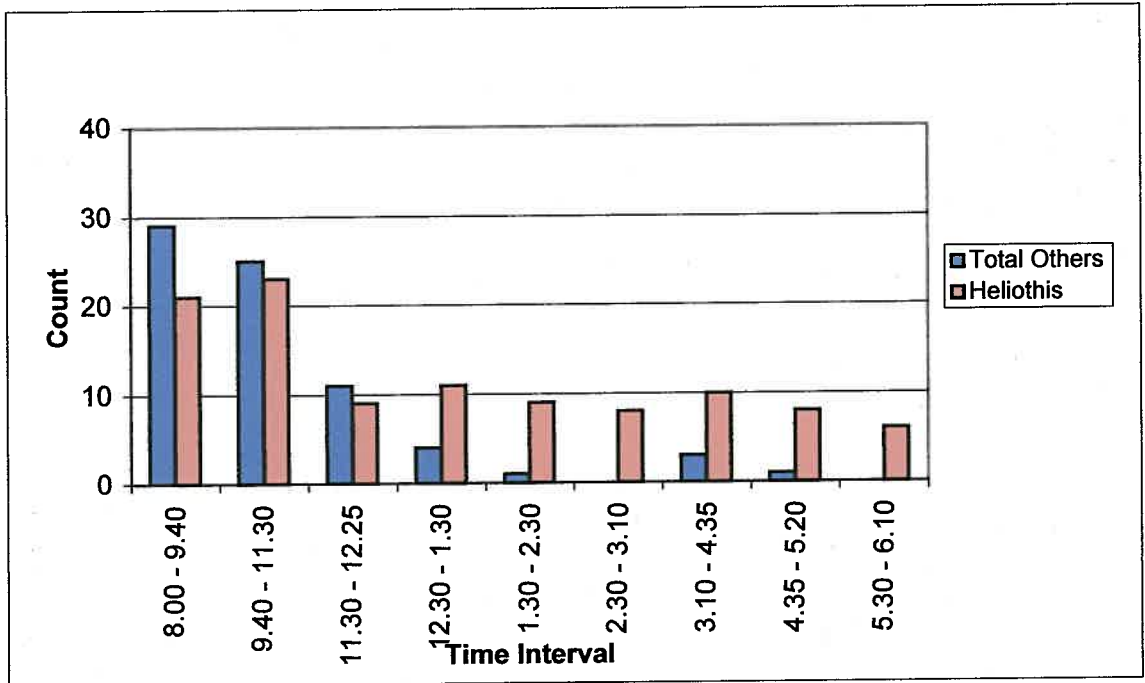


Figure 12. Light trap Yarral 22-23/2/2000 Total *Helicoverpa* moths captured per time interval

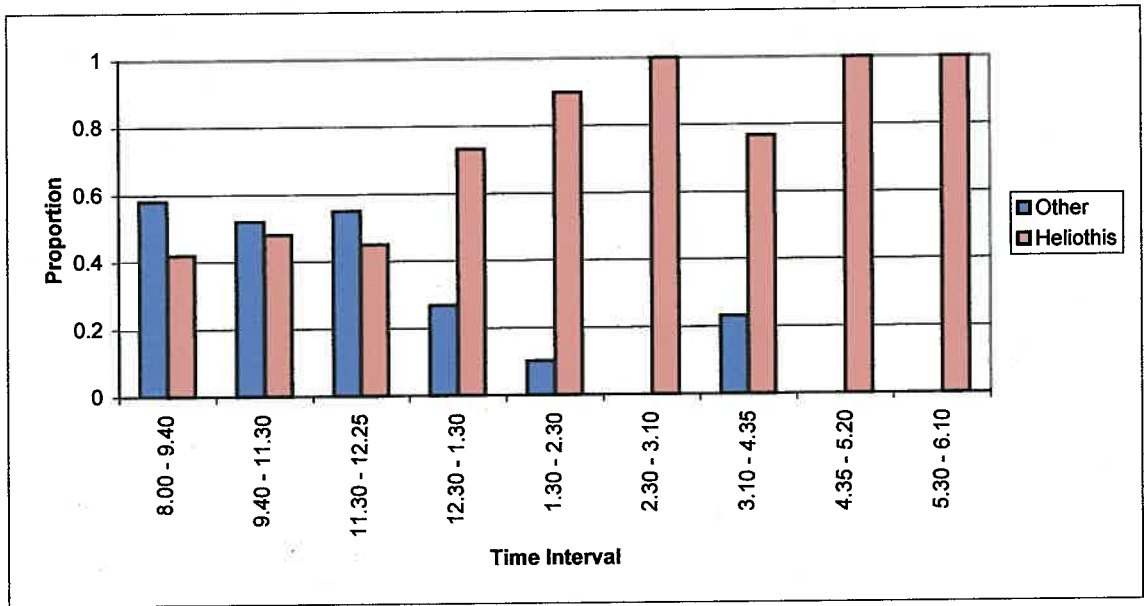


Figure 13. Light trap Yarral 22-23/2/2000 Proportion *Helicoverpa* moths

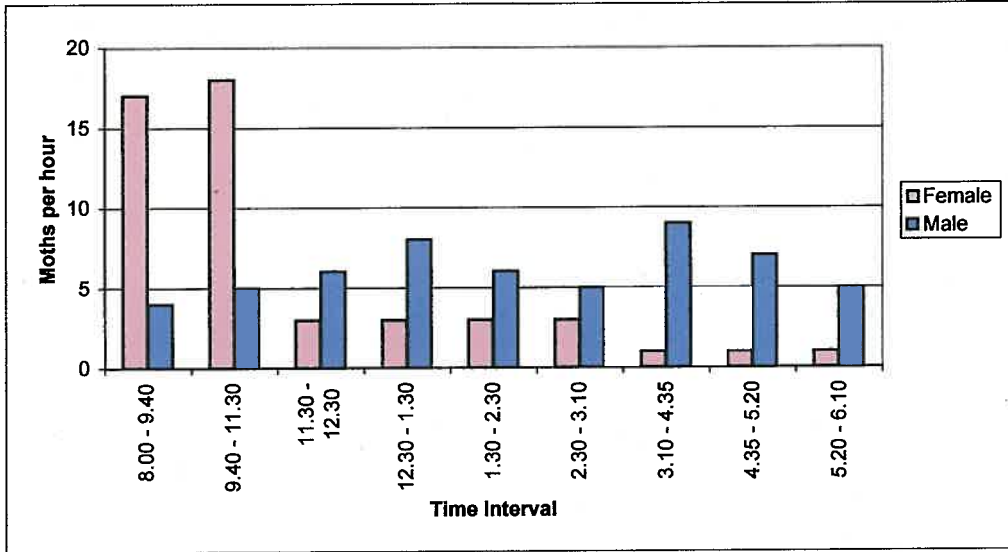


Figure 14. Light trap Yarral 22-23/2/00 *Helicoverpa* flight activity per hour during each time interval.

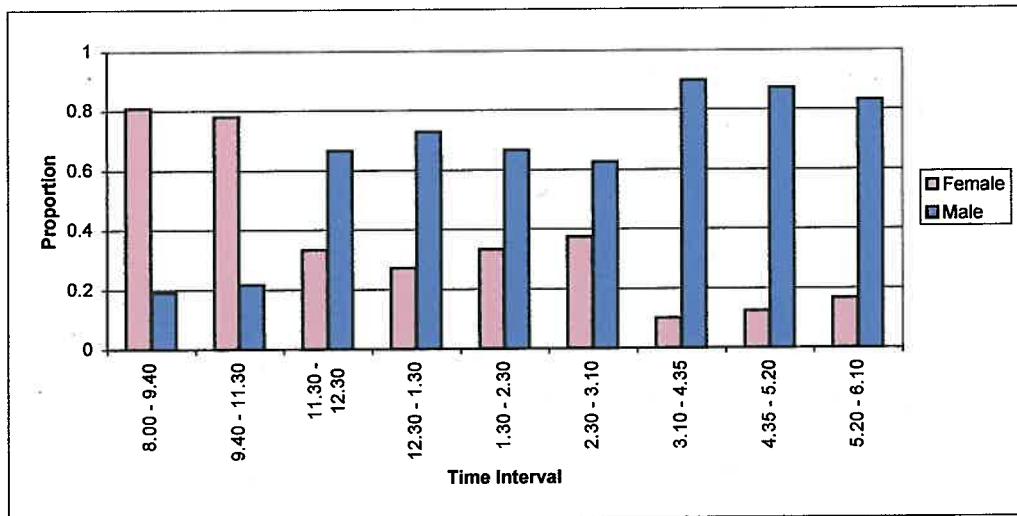


Figure 15. Light trap Yarral 22-23/2/2000 *Helicoverpa* sex ratio per hour during each time interval.

#### *The timing of Helicoverpa egg laying activity.*

The field plant exclusion experiments (photo 15) designed to quantify the nocturnal timing of *Helicoverpa* egg laying did not produce any consistent data due to insufficient and irregular egg pressure. However the laboratory experiments were successful, and the results of two of these trials are presented in Figures 16a and 16b. Pilot experiments had indicated that the captive moths were potentially disturbed by the hourly interruptions to count eggs. For this reason two experiments were conducted. In trial 1 (Figure 16a), the eggs were checked at hourly intervals for the first 4 hours of darkness, and then were left undisturbed for the remainder of each night. In trial 2 (Figure 16b) this was reversed, and the moths were left undisturbed for the first 4 hours, and were then checked at hourly intervals to count eggs laid. Combining the results of the two experiments suggests that in these laboratory experiments, female *Helicoverpa* moths laid eggs throughout the night at a relatively uniform rate. There were no hourly periods that received significantly more or less eggs than other periods.

#### *The spatial distribution of Helicoverpa larvae within a trap crop.*

Figure 17 provides a shaded representation of the density of *Helicoverpa* larvae recorded by 480 beat sheet samples in the 5 Ha block of pigeon pea at Lowana. In all 2177 larvae were collected. The larvae were not distributed uniformly within the crop. There was a strong trend of higher densities in the southern end of the trap crop. However no measurable agronomic or physical reasons for this difference were readily evident, and the increase in density did not appear to be related to the edge of the trap crop per se. The average density around the sample points closest to the edge was 2.94 larvae/m compared with the average density of the central sample points of 2.72 larvae/m. The pigeon pea trap crop was surrounded by cotton on all sides. Collections of larvae were not completed in adjacent cotton on this date, and the distribution of *Helicoverpa* larvae in the surrounding cotton crops is unknown.

#### *The timing of Helicoverpa pupal diapause relative to trap crop phenology.*

HEAPS model predictions of the timing of the cumulative spring emergence of *H.armigera* moths from their over-wintering diapause were used in the Spring Trap Crop Management Guidelines (Appendix 5). Predictions were also produced on request for other cotton researchers and Industry Development Officers. Figure 18 is an example of the model output predicting the timing of emergence over 12 years at Griffith in southern NSW based on historical weather data. This type of information is useful for fine-tuning the planned flowering dates of spring trap crops to maximise their effectiveness at capturing the first generation of *H.armigera*. Similarly, Figure 19 provides examples of HEAPS predictions of autumn diapause induction at Griffith. The information is important for farmers planning to destroy and cultivate late summer trap crops (and cotton and other crop residues) at the end of the season.

The APSIM model predictions of the probable flowering times of chickpea trap crops at a range of locations and under full and half moisture profiles were incorporated into the Spring Trap Crop Management Guidelines (Appendix 5). An example of the detailed output of the model results for Boggabri in the upper Namoi is presented in Table 3a and 3b.

**Table 3a.** APSIM Predicted flowering dates based on long term temperatures for Sorghum (early maturing) sown at Boggabri, NSW on full moisture profile.

Sowing Date	80% probability for start of flowering	Average date for start of flowering	20% probability for start of flowering
2-Oct	17-Dec	22-Dec	26-Dec
9-Oct	22-Dec	27-Dec	2-Jan
16-Oct	27-Dec	2-Jan	6-Jan
23-Oct	1-Jan	6-Jan	11-Jan
30-Oct	7-Jan	12-Jan	17-Jan
6-Nov	13-Jan	18-Jan	23-Jan
13-Nov	19-Jan	24-Jan	29-Jan
20-Nov	25-Jan	30-Jan	3-Feb
27-Nov	31-Jan	5-Feb	10-Feb
4-Dec	7-Feb	12-Feb	16-Feb
11-Dec	13-Feb	18-Feb	22-Feb
18-Dec	20-Feb	24-Feb	1-Mar
25-Dec	27-Feb	3-Mar	8-Mar
1-Jan	7-Mar	10-Mar	14-Mar

**Table 3b.** APSIM Predicted flowering dates based on long term temperatures for Sorghum (early maturing) sown at Boggabri, NSW on half moisture profile.

Sowing Date	80% probability for start of flowering	Average date for start of flowering	20% probability for start of flowering
2-Oct	22-Dec	3-Jan	10-Jan
9-Oct	27-Dec	7-Jan	14-Jan
16-Oct	2-Jan	12-Jan	19-Jan
23-Oct	6-Jan	16-Jan	24-Jan
30-Oct	12-Jan	22-Jan	31-Jan
6-Nov	17-Jan	26-Jan	3-Feb
13-Nov	23-Jan	31-Jan	7-Feb
20-Nov	28-Jan	5-Feb	12-Feb
27-Nov	31-Jan	5-Feb	10-Feb
4-Dec	10-Feb	20-Feb	25-Feb
11-Dec	16-Feb	25-Feb	4-Mar
18-Dec	23-Feb	5-Mar	10-Mar
25-Dec	2-Mar	11-Mar	17-Mar
1-Jan	8-Mar	18-Mar	23-Mar

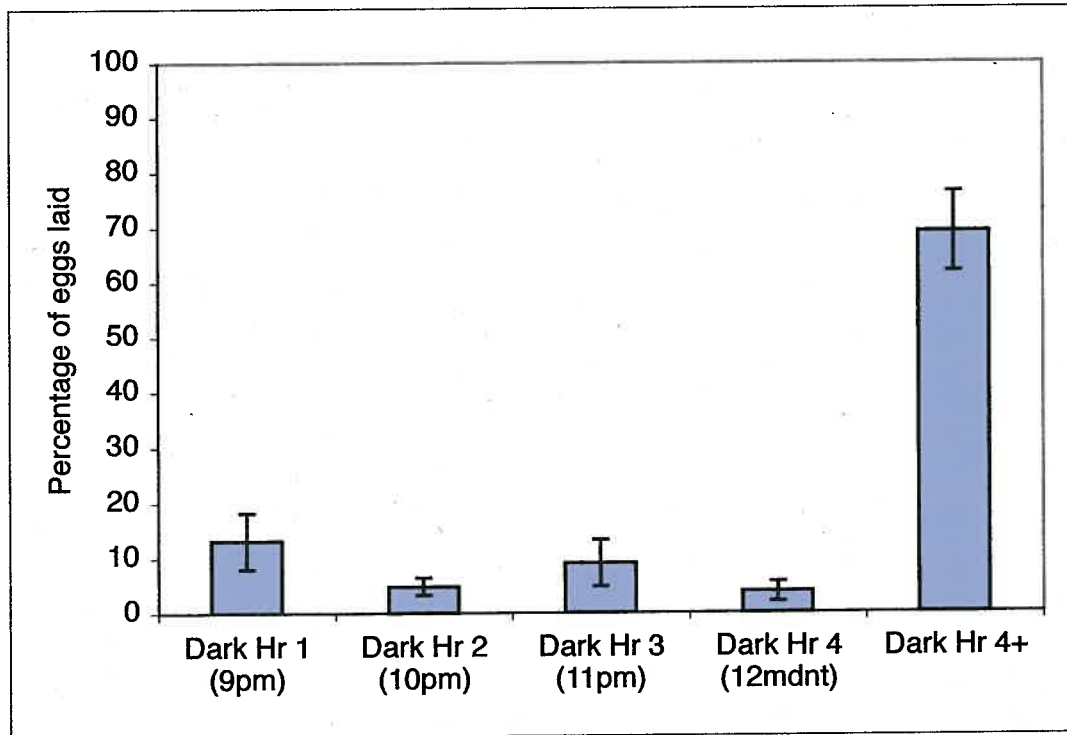


Figure 16(a). Timing of egg lays in laboratory experiment 1

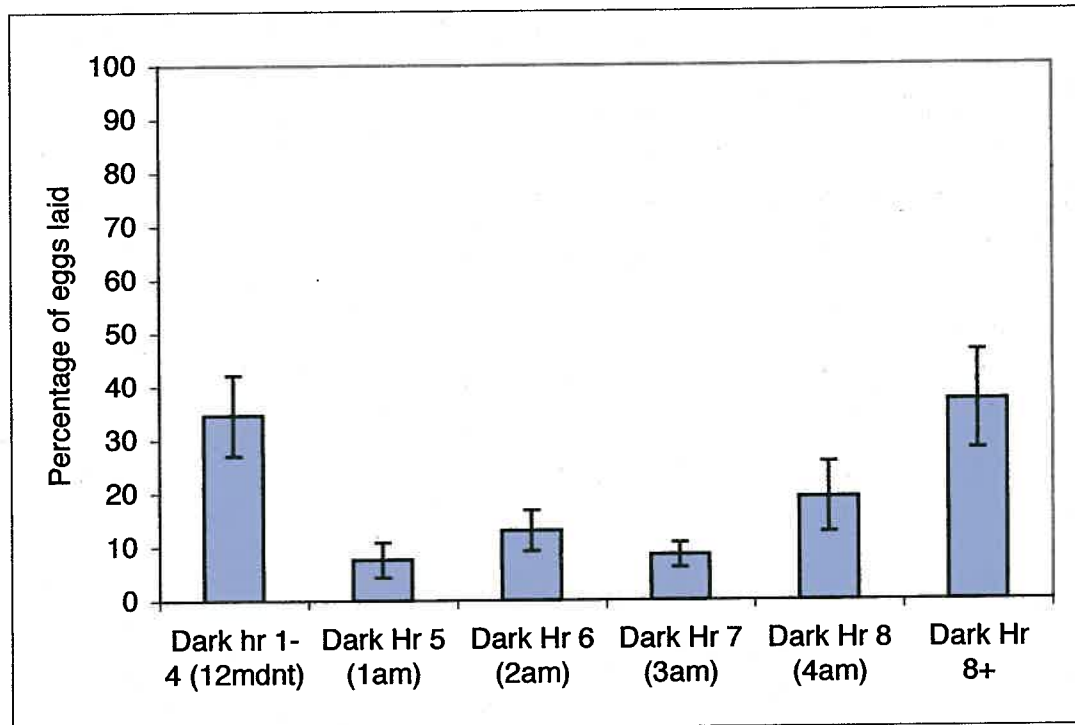
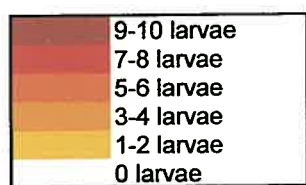
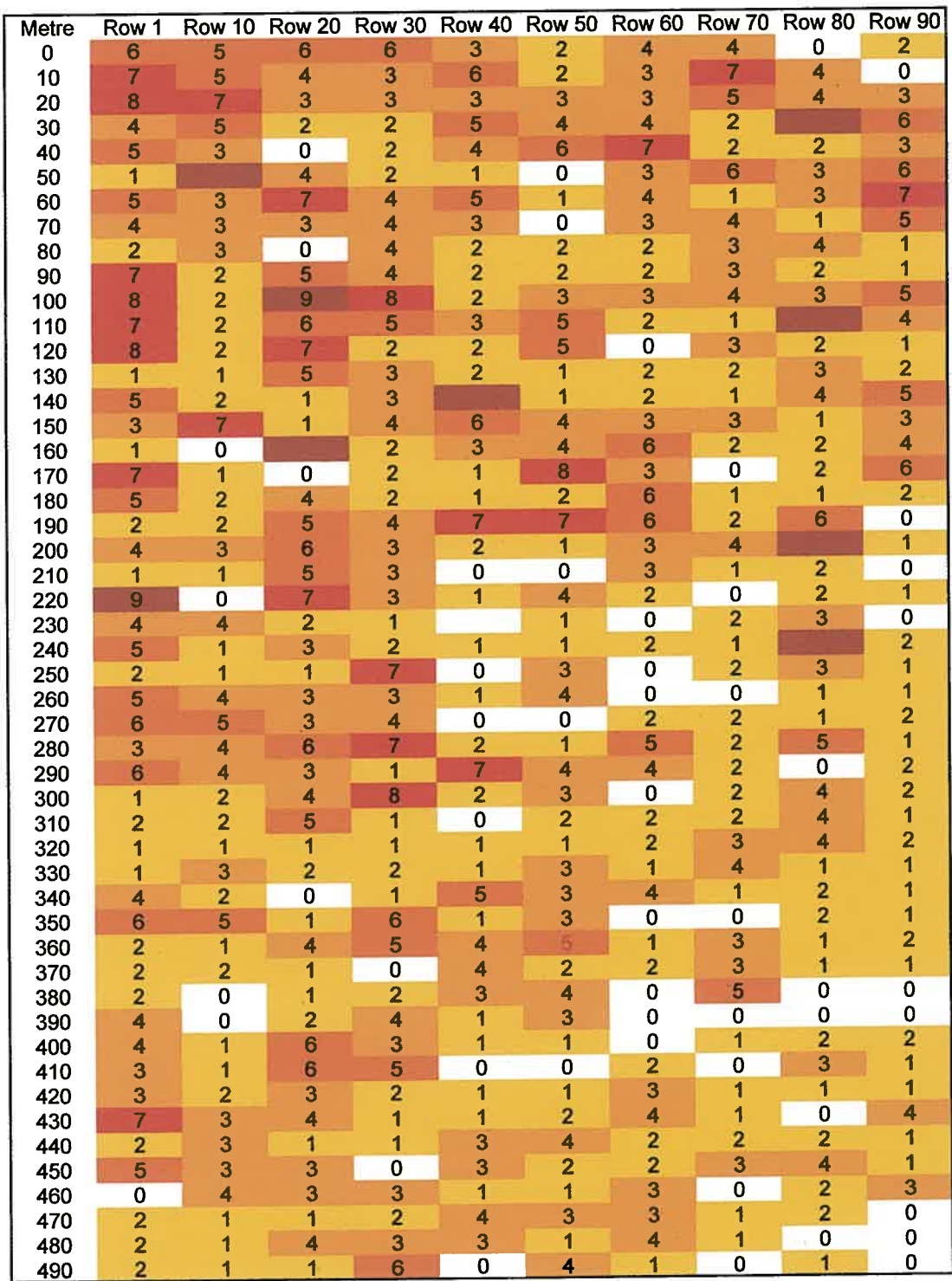


Figure 16(b). Timing of egg lays in laboratory experiment 2



North

Figure 17. *Helicoverpa* larvae density in Pigeonpea trap crop 150 x 500 metres



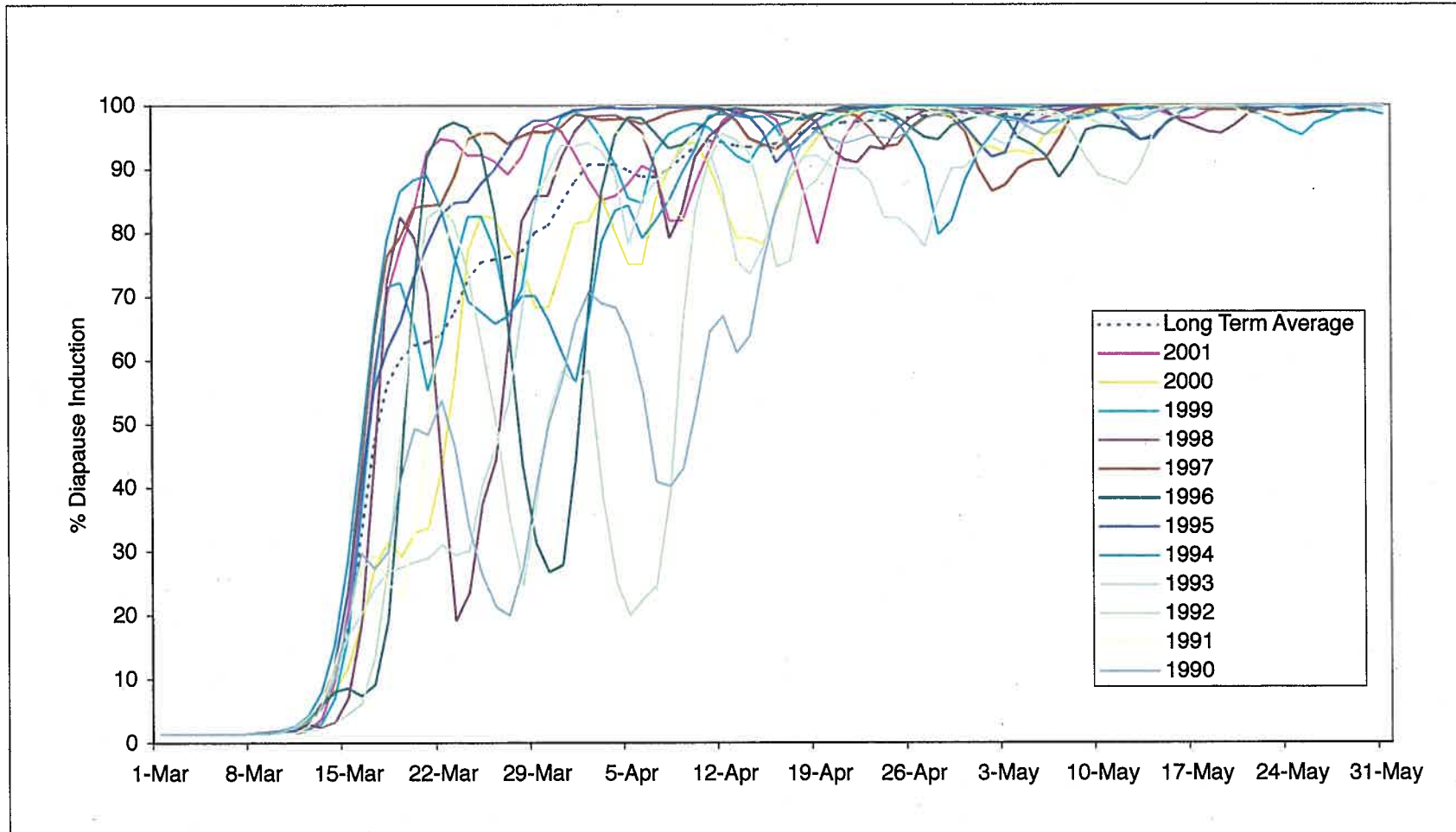
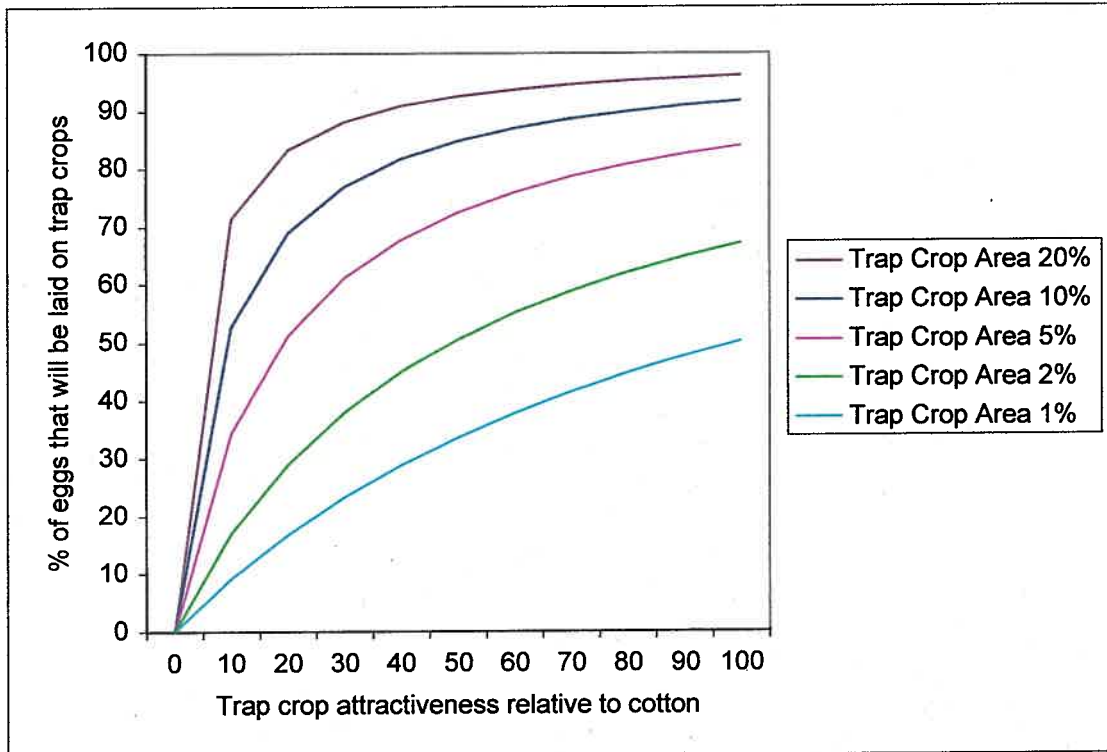


Figure 19. HEAPS simulation: *H. armigera* Cumulative % Autumn Diapause Induction at Griffith in NSW over 12 years.

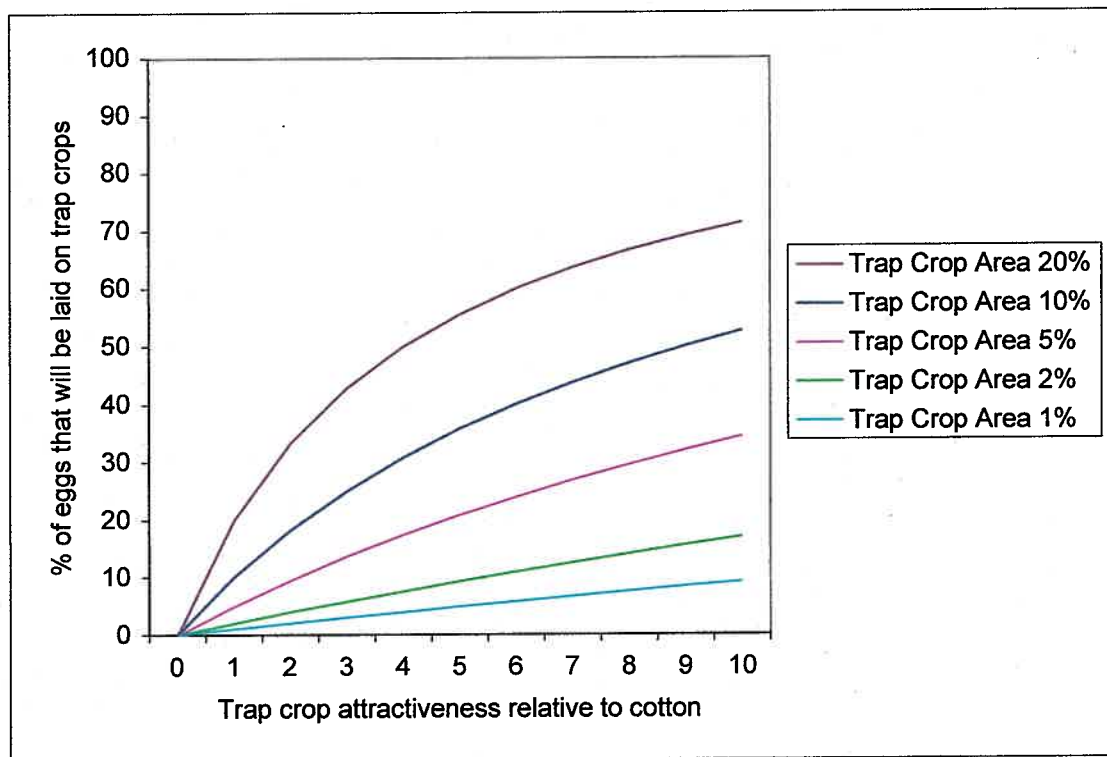
*The proportion of cropping area of trap crops required for effective operation.*

Most trap cropping programs within the Australian cotton industry have been implemented on the basis of planting about 1% of the cropping area on a farm or within an Areawide management group to trap crops. During early spring and late summer when trap crops are not competing with cotton as hosts for *Helicoverpa* eggs, a 1% area may well provide a very effective population sink. However for trap crops designed to divert eggs away from cotton there is no scientific data available to determine the optimal proportional area of trap crops. There is also a lack of specific data quantifying the relative attractiveness of different types of host crops to *Helicoverpa* under commercial farming conditions. However some estimates can be made of the likely effectiveness of different proportional areas of trap crop using a range of assumptions about relative attractiveness and moth dispersal behaviour. Spreadsheet model predictions of the likely proportion of *Helicoverpa* eggs within a discrete region that can be expected to be laid onto trap crops are presented in Figures 20 and 21. For 5 different trap crop areas (1%, 2%, 5%, 10% and 20%), the predicted proportion of total *Helicoverpa* eggs that will be laid onto trap crops within the landscape is predicted for a range of trap crop attractiveness relative to cotton. The attractiveness scale can be interpreted as the number of eggs that would be laid onto the trap crop, for every 1 egg laid onto cotton if moths had a clear choice.

The results in Figure 20 clearly show that for a trap crop competing against cotton, either the trap crop must be highly attractive (tens of times more attractive than cotton), or there must be a substantial area of trap crop in the landscape for an appreciable proportion of eggs to be diverted away from cotton. Figure 21 presents the same data, but at a finer scale, with relative attractiveness ranging from 1 to 10. It is likely that most trap crops used for *Helicoverpa* would have levels of attractiveness in this range. The results suggest that for trap crops that compete against cotton, if the total area set aside for trap crops is less than 2%, then the proportion of *Helicoverpa* eggs that could be expected to be laid onto trap crops is about 10% or less. It must be emphasised that these are theoretical predictions, and do not consider potential aspects of *Helicoverpa* behaviour that may influence how and where they lay their eggs. For example, the work of Dr Paul Cunningham and Prof. Myron Zalucki suggests that moths may 'learn' to lay eggs on the host that they encounter most and are familiar with.



**Figure 20.** The proportion of eggs that would be laid on trap crops over a range of attractiveness (1 to 100x) relative to cotton for 5 different trap crop areas.



**Figure 21.** The proportion of eggs that would be laid on trap crops over a range of attractiveness (1 to 10x) relative to cotton for 5 different trap crop areas.

## 5. Conclusions and 'take home messages'

- Spring trap crops are effective at diverting eggs away from young cotton.
- Spring trap crops can attract very high densities of eggs, and therefore provide a significant population sink for the first generations of *Helicoverpa* emerging from diapausing pupae or arriving as immigrants.
- A brochure "Spring Trap Crop Management Guidelines" produced in collaboration with DPI researchers provides an overview of the benefits of spring trap crops and recommendations for agronomic management.
- In-season trap crops of Pigeonpea are only highly attractive for short periods (~7 days) immediately prior to, and during, initial flowering. Results were variable, but in general flowering pigeon pea subsequently attracted similar densities of eggs as those present in nearby cotton.
- A staggered planting of three blocks of pigeon pea that commenced flowering at three week intervals resulted in a relatively continuous high level of attractiveness relative to cotton.
- Sorghum trap crops are highly attractive as the flower first emerges from the boot for a period of 7 to 10 days.
- Late season trap crops of sorghum (especially staggered plantings) are highly attractive relative to mature cotton at that time, and can attract extremely high densities of eggs.
- Late season trap crops of sorghum have the potential to provide a significant population sink for the last generation of *Helicoverpa*. They attract eggs that would otherwise be laid elsewhere on the landscape and hatch into individuals that would later become diapausing pupae.
- Trap crops in a block configuration are superior to strips or patches, and some data suggests that strips are superior to small patches per unit area.
- As the size of an individual block of trap crop increases, the egg densities per unit area that it attracts also increases. The reasons for this are unknown. It may be that the perimeter to area ratio falls as trap crop size increases, or it may be attributable to aspects of moth behaviour.
- No evidence of egg density gradients were detected in cotton surrounding trap crops.
- Spreadsheet modelling studies suggest that for trap crops to effectively compete directly against cotton, they must either be highly attractive (in the order of 10 or more times more attractive than cotton) and/or the proportional area of the landscape planted to trap crops must be relatively high. For example, if a trap crop is 10 times more attractive than cotton, and represents 2% of the cropping area, it is predicted that about 15% of all *Helicoverpa* eggs will be laid onto the trap crop, and 85% will still be laid onto cotton. Note that these results are not applicable in early spring or late summer when trap crops may not be 'competing' with cotton.
- Trap crops are compatible with and complimentary to INGARD® refuges. This was a confusing issue for many cotton growers and consultants. A cotton grower article was published outlining how trap crops and refuges work (Appendix 9).

## 6. Economic, Environmental and Social outputs of this research

This research is relevant to the sustainability and profitability of the cotton industry because it examined aspects related to the deployment and impact of trap crops for attracting *Helicoverpa* eggs. Results indicate that spring trap crops (especially chickpea) can be very successful, and have the potential to substantially impact on the first spring generation - thereby suppressing subsequent populations, and possibly allowing a significant proportion of the regional population to be controlled with 'soft' chemicals and/or mechanical means. Such a suppression of a regional pest population has the potential to reduce overall chemical use and to help conserve susceptible genes in the pest population. The efficacy and potential benefits of in-season summer trap crops were more variable, and sometimes eggs were only diverted away from cotton for only short periods during initial flowering. Late season trap crops were very effective at attracting high densities of *Helicoverpa*. These represent the last generation of predominantly *H.armigera* - the survivors and progeny of survivors of the whole seasons insecticide regime. These individuals are likely to carry resistance genes, and late season trap crops provide an effective and sustainable means of cultural control.

A direct outcome of this work has been Mr Dillon's significant contribution to the "Spring trap crop guidelines" brochure published in August 2000. The project has addressed the people and communities outcome by providing advisory support to new and existing areawide management groups that are deploying trap crops and/or implementing other IPM practices in the Macquarie, Namoi, Gwydir and Macintyre valleys. The research completed during this project underpins progress towards an increased adoption of IPM approaches, and a reduction in cotton growers' dependence on chemical insecticides.

## 7. Summary

### a) Technical Advances

The major technical result to arise from this work has been the clear demonstration that *Helicoverpa* populations can be manipulated to lay their eggs onto trap crops within the landscape. This is particularly so during spring and late summer when cotton is either not present or not attractive as an oviposition host.

The project also clearly demonstrated that the spatial configuration of individual trap crops influences their attractiveness. Larger trap crops attract proportionally more eggs, and blocks attract more eggs than strips or patches. These findings have been incorporated into the Ingard® resistance management strategy by stipulating the minimum dimensions of refuges.

### b) Other Information Developed

A number of methods for studying *Helicoverpa* behaviour and ecology were developed in the course of this research. In particular, the use of night vision goggles, infra-red spotlights and goal posts has been a novel approach that allows comparison and quantification of moth activity levels within specific crops.

### c) Changes to the Intellectual Property Register

None required. There were no commercially significant innovations achieved during this project.

## 8. Future plans:

### a) *Further Development of Project Technology*

The utilisation of trap crops within areawide management groups continues to be evaluated as one component of M.Dillon's current CRDC project CSE103C "The impact of area wide management on beneficial arthropod and *Helicoverpa* populations".

Methods for quantifying moth flight behaviour and activity levels over crops continue to be refined.

### b) *Presentation and Dissemination of Project Outcomes*

Project results have been extensively presented to growers, consultants and scientists. See section 9 below for a complete listing. Future planned scientific manuscripts are also listed in section 9(f) below.

### c) *Future Research*

The recent registration of the "Magnet" *Helicoverpa* attractant, the ongoing research and development into *Helicoverpa* repellents and trap-based suppression systems by commercial companies, and advances in scientific understanding of *Helicoverpa* chemosensory ecology and host preference and learning behaviour all have substantial implications for trap crop research. Such technologies that manipulate, or 'push' and 'pull', populations of *Helicoverpa* around the agricultural landscape are likely to be fertile areas for future research that may lead to sustainable management systems for this major pest.

## 9. Publications Arising and Planned

### a) Industry Talks, Education, Training and Media

Research findings have been presented at field days, farm walks and shed meetings at Narrabri, Pilliga, Wee Waa, Boggabri, Moree, Warren, Tandou, Walgett, Bourke, Boggabilla, Goondiwindi, and Dalby.

Over the course of the project advice on deploying and managing spring and summer trap crops has been given to cotton Industry Development Officers, Areawide Management groups, and numerous individual cotton growers.

Each year since 1999 a lecture on "Trap Cropping" has been presented to UNE Cotton Production Course residential students visiting ACRI, Narrabri.

Mr Dillon presented reviews of the trap crop project at the Toowoomba Grains IPM workshop (June 2001), and at the Total Ag 'IPM Day' (November 2001). Mr Dillon gave two presentations to the Lower Namoi Pilot IPM Short course.

A research overview seminar was presented at a Certification & Up-skilling Workshop for Cotton Consultants of Australia Inc., Wee Waa, NSW. 12 September 2002.

M.Dillon was filmed for two "Web on Wednesday" online video interviews broadcast on the CSD website: 'Late season trap crops and compatibility with Ingard® refuges' (8 November 2001); and, 'Heliothis predictions' (9 October 2002).

M.Dillon was interviewed for three articles published in 'The Land' national newspaper:  
'Heliothis weapon in chickpeas' (15 July 1999).  
'Trap crops value unknown' (22 February 2001).  
'Trap crops for heliothis' (19 September 2002).

#### b) Industry Guidelines

Dillon ML. 1999. *Helicoverpa armigera* diapause induction and emergence tables. Support Document 2 in Mensah R and Wilson L. (Editors). "*Integrated Pest Management Guidelines for Australian Cotton*", Australian Cotton Cooperative Research Centre, Narrabri. 1999. [Appendix 1]

Mensah R, Dillon ML, Khan M, Tann C and Wilson L. 1999. Cotton Insect Pests and their crop hosts. Support Document 3 in Mensah R and Wilson L. "*Integrated Pest Management Guidelines for Australian Cotton*", Australian Cotton Cooperative Research Centre, Narrabri. 1999. [Appendix 2]

Mensah R, Dillon ML, Khan M, Tann C and Wilson L. 1999. Cotton Insect Pests and their weed hosts. Support Document 4 in Mensah R and Wilson L. "*Integrated Pest Management Guidelines for Australian Cotton*", Australian Cotton Cooperative Research Centre, Narrabri. 1999. [Appendix 3]

Dillon ML. 1999. Guide to planting dates and flowering dates for chickpea trap crops. Support Document 6 in Mensah R and Wilson L. "*Integrated Pest Management Guidelines for Australian Cotton*", Australian Cotton Cooperative Research Centre, Narrabri. 1999. [Appendix 4]

Ferguson J, Miles M, Murray DAH, Dillon ML, Kauter G, and Lloyd R. 2000. *Spring Trap Crop Management Guidelines*. Australian Cotton Cooperative Research Centre, Cotton Research and Development Corporation and Grains Research and Development Corporation.

[Appendix 5]

Wilson, L.J., Mensah R., Dillon M., Wade M., Scholz B., Murray D.A., Heimoana V., and Lloyd R. (2002) Impact of insecticides and miticides on predators in cotton: October 2002 update. Support Document 1 in: Mensah, R. and Wilson, L.J. (Editors): *Integrated Pest Management Guidelines for Australian Cotton*. Australian Cotton Cooperative Research Centre, Narrabri, NSW. <http://cotton.pi.csiro.au/Publicat/Pest/> [Appendix 6]

Hickman M, Larsen D and Dillon ML. 2002. Guidelines for assessing pupae risk for dryland cotton growers. Cotton Information Sheet July 2002. Australian Cotton Cooperative Research Centre, Narrabri. [Appendix 7]

c) Conference and Seminar Presentations

Dillon ML. 1999. Trap Cropping for *Heliothis*. Paper presented to the CSD/CSIRO Cotton Research Review, 11-12 May 1999 Narrabri.

Dillon ML 1999. Trap cropping attractiveness and performance. Invited presentation. Proceedings of the Cotton Production Seminar (Cotton Consultants Australia Inc.), Goondiwindi 26-27 August 1999. [Appendix 8]

Dillon, ML.. 2000. Trap crops for *Helicoverpa* in Australian cotton systems. Oral Presentation at the Australian Entomology Conference, Darwin, June 2000.

Fitt, GP., and Dillon, ML. 2000. Cropping patterns: A landscape perspective of cultural control. Oral Presentation for the International Congress of Entomology, Igguasu Falls, Brazil. in the Agricultural Entomology symposia: "New wine from old skins: A landscape view of cultural control".

Dillon, ML., Fitt, GP. 2000. *Heliothis* local movement and resistance management for transgenic cotton. Oral Presentation for the International Congress of Entomology, Igguasu Falls, Brazil. Presented in the symposia: "Forecasting and managing migratory insect pests and natural enemies".

Dillon ML. 2001. Areawide Management in Northern NSW. Paper presented to the CSD/CSIRO Cotton Research Review, June 2001 Narrabri.

Dillon M. 2003. IPM on an Areawide Scale in the Macintyre Valley. Proceedings of the Cotton Production Seminar (Cotton Consultants Australia Inc.), Narrabri 26-27 August 2003.

Dillon ML. 2003. Understanding the components of successful areawide management. Presentation to Cotton CRC Annual Review, Armidale, 23-24 July 2003.

Dillon ML. 2003. Research Overview presented at "DPI Farming Systems IPM Meeting" Toowoomba, 25/26 June 2003.

M.L. Dillon. 2003. Trap crops for managing *Helicoverpa* (Lepidoptera: Noctuidae) in Australian cotton farming systems. Oral presentation at the World Cotton Research Conference-3, Cape Town, South Africa (March 2003).

M.L. Dillon. 2004. Invited Presentation (2004, Forthcoming) Trap crops for managing *Helicoverpa armigera* in Australian cotton. Paper presentation in Cotton Pest Management Symposium, XXII International Congress of Entomology, Brisbane, Queensland, Australia.

d) Industry Publications

Fitt, GP.; Dillon ML and Tann, C. 2000. Entomological Research Update: Cotton Season 1999-2000. Pages 84-85 in Cotton Seed Distributors 2000 Variety Trial Results & Grower Information. CSD, Wee Waa.

Dillon M. and Hoque Z. 2000. An analysis of pest pressure within an areawide management group. Pages 75-83 in Proceedings of the 10th Australian Cotton Conference, Brisbane, Qld. 16-18 August 2000.

Dillon ML. 2001. INGARD® Refuges v Trap Crops: Are they compatible? *The Australian Cottongrower* 22(6): 10-12. [Appendix 9]

Dillon ML, Tann, C. and Baker G. 2001. Entomological Research Update: Cotton Season 2000-2001. Pages 90-91 in Cotton Seed Distributers 2001 Variety Trial Results & Grower Information. CSD, Wee Waa.

Dillon ML and MacKinnon L. 2002. Using light traps to suppress *Helicoverpa*. *The Australian Cottongrower* 23(2): 32-36.

Tann C., Dillon ML and Baker, G. 2002. Entomological Research Update: *Helicoverpa* Cotton Season 2001-2002. Pages 102-103 in Cotton Seed Distributers 2002 Variety Trial Results & Grower Information. CSD, Wee Waa.

e) Scientific Manuscripts

M.A.Merritt, K.S.Wilkinson, K.D.Scott, M.Dillon, L.J.Scott, C.L.Lange, M.K.Schutze, J.K.Kent, D.J.Merritt And G.C.Graham. (2004, submitted.) Gene flow in two consecutive collections of the Lepidopteran pest, *Helicoverpa armigera* (Hübner) (Lepidoptera: Noctuidae) from the Narrabri region, New South Wales. Australian Journal of Agricultural Research.

Heuke L and Dillon ML. (In preparation) Growing pigeon pea as a refuge and trap crop for cotton. [Appendix 10]

Duffield SJ and Dillon, ML. (In preparation). The emergence and control of overwintering *Helicoverpa armigera* in southern NSW. [Appendix 11]

f) Planned Scientific Manuscripts

“Trap crops for *Helicoverpa* spp. in Australian cotton production”.

- This paper will present an overview of trap cropping as an IPM tool for managing *Helicoverpa* spp in Australian cotton production systems. It will review farm level and AWM group case studies of the deployment of trap crops. A synopsis of this paper has been accepted for presentation as an invited talk at the forthcoming XXII International Congress of Entomology in Brisbane, 15-21 August 2004. The full paper will be submitted for internal review in July 2004 and will be targeted at the international journal “Environmental Entomology”.

“Trap crop algebra: the theoretical effectiveness of trap cropping strategies deployed against *Helicoverpa* moths”.

- This paper will present the results of modelling studies on the effectiveness of a range of trap crop deployment strategies against *Helicoverpa*. Sensitivity analysis will be used to examine how a range of likely moth flight and oviposition behaviours interact with the spatial layout, size, floral timing and attractiveness of trap crops. The paper will be submitted for internal review in August 2004 and will be targeted at the Australian Journal of Entomology.

“Estimating *Helicoverpa* moth densities over cotton crops using night-time goal post counts”.

- This paper will present results of modelling studies of moth flight behaviour validated against visual observations of nocturnal and diurnal moth counts and white egg densities. . The paper will be submitted for internal review in September 2004 and will be targeted at the Australian Journal of Entomology.

“Optimal flight strategies for moths in relation to wind speed, wind direction and the distribution of host patches in agro-ecosystems”.

- This paper will present a theoretical analysis demonstrating that the optimal flight strategy for a moth seeking suitable host patches in a diverse landscape is to fly directly downwind. Such behaviour maximises groundspeed per unit of expended energy, thereby increasing the probability of encountering suitable host patches per unit time with minimum effort. The paper will discuss the implications of agricultural landscapes that are restricted to fertile ‘valleys’ and how this may explain some of the differences in observed dispersal between *H.armigera* which has evolved in close association with human agriculture, and *H.punctigera* an endemic Australian moth adapted to utilising ephemeral host patches in a large scale arid landscape. The paper will be submitted for internal review in October 2004 and will be targeted at an international entomological journal.

## 10. Impact of project Results and Conclusions for the Cotton Industry

The findings of this project confirm that spring and late summer trap crops can play a very valuable role in providing a significant population sink for *Helicoverpa*. In doing so, trap crops help to suppress the overall population, and possibly subsequent populations, of both species of *Helicoverpa* within a region. Such suppression must lead to reduced egg pressure on cotton crops, and this potentially reduces the frequency and/or severity of chemical insecticides that farmers may otherwise have had to apply to protect their crop. By reducing farmers’ dependence on chemical insecticides, trap crops help to slow the evolution of resistance, and they help to reduce potential adverse environmental effects of disruptive sprays.

The project findings suggest that mid-summer trap crops like pigeon pea are not always highly attractive, and may not necessarily divert eggs away from cotton. The demonstration of a significant influence of individual trap crop spatial configuration on the number of eggs attracted, and the apparent lack of evidence of gradients in *Helicoverpa* egg density in proximity to cotton crops suggests that there are aspects of moth egg laying behaviour that we do not yet fully understand or know about. The theoretical evaluation of the proportion of the cropping area that should be sown to trap crops demonstrated that if trap crops have to compete against cotton they need to be either highly attractive and/or planted in proportionally large areas.

Some of the information on the effectiveness at attracting eggs of individual trap crop size and layout configurations has been utilised in the INGARD® Resistance Management Strategy. In particular, the minimum requirements for the size and shape of INGARD® refuges takes into account the finding that patches and strips are inferior to blocks.

## 11. Acknowledgements

Trudy Staines and Louise Munday provided exceptional technical support both in the field and laboratory. Thanks also to the two enthusiastic summer students Sydney Jordan and Sabina Kindler, and to the following casual assistants: Mary Fielder, Tanya Nolan, Tracey Parker, Karen Stanford, Colin Tann, Debbie Michelle, Patrick Halloway, Leah MacKinnon, and Jenny Chapman. Sincere thanks are extended to Ken and Lou Platt for their generous support and hospitality in allowing trap crop research to be undertaken on their property 'Lowana', Pilliga NSW. Thanks to the owners and staff of 'Auscott' 'Yarral' and 'Myambla'. Thanks to the farm staff of CSIRO and Agriculture NSW at the Australian Cotton Research Institute for assisting with trap crop and cotton trials.

## ***Part 4 – Final Report Executive Summary***

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Trap crops act as diversionary hosts for key pests, attracting and concentrating the pests and their eggs into a relatively small area where the population can be controlled, often by mechanical means. Within the Australian cotton industry trap crops are planted by individual farmers, and/or by members of Areawide management groups as a means of diverting *Helicoverpa armigera* and *H.punctigera* eggs away from cotton and also to suppress the overall population of *Helicoverpa* within a locality.

This project set out to quantify the effectiveness of the main types of trap crops deployed by cotton farmers, and to measure their impact on egg densities in surrounding cotton. Another aim was to quantify the night-time activity levels and egg laying behaviour of *Helicoverpa* moths within trap crops and in associated cotton crops.

In the course of the project over 50 trap crops were monitored and sampled. The main trap crops studied were chickpeas, pigeon peas, sorghum, and sunflower. The findings of this project confirmed that spring and late summer trap crops can play a very valuable role by attracting very high densities of *Helicoverpa* eggs and therefore in creating a significant population sink. In doing so, trap crops help to suppress the overall population, and possibly subsequent populations, of both species of *Helicoverpa* within a region. Such suppression must lead to reduced egg pressure on cotton crops, and this potentially reduces the frequency and/or severity of chemical insecticides that farmers may otherwise have had to apply to protect their crop. By reducing farmers' dependence on chemical insecticides, trap crops help to slow the evolution of resistance, and they help to reduce potential adverse environmental effects of disruptive sprays.

The project findings suggest that mid-summer trap crops like pigeon pea are not always highly attractive, and may not necessarily divert eggs away from cotton. The failure to detect any evidence of gradients in *Helicoverpa* egg density in proximity to cotton crops, and the significant influence of individual trap crop spatial configuration on the number of eggs attracted suggests that there are aspects of moth egg laying behaviour that we do not yet fully understand or know about. The theoretical evaluation of the proportion of the cropping area that should be sown to trap crops demonstrated that if trap crops have to compete against cotton they need to be either highly attractive and/or planted in proportionally large areas.

The project demonstrated that larger individual trap crops attracted proportionally higher densities of eggs than smaller trap crops. Direct comparisons showed that trap crops planted as blocks attracted significantly higher egg densities than strips or patches. Some of the information on the effectiveness at attracting eggs of individual trap crop size and layout configurations has been utilised in the INGARD® Resistance Management Strategy. In particular, the minimum requirements for the size and shape of INGARD® refuges takes into account the finding that patches and strips are inferior to blocks.

During the project M.Dillon made a substantial contribution to the production of the "Spring Trap Crop Management Guidelines" for cotton growers. Project results were also widely disseminated at field days, farm walks, industry seminars and conferences, and in industry related print media.

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## Appendix 1

Dillon ML. 1999. *Helicoverpa armigera* diapause induction and emergence tables. Support Document 2 in Mensah R and Wilson L. (Editors) "*Integrated Pest Management Guidelines for Australian Cotton*", Australian Cotton Cooperative Research Centre, Narrabri. 1999.

# *Helicoverpa armigera* diapause induction and emergence tables

Prepared by Martin Dillon<sup>1</sup>

## What is diapause ?

The pupal stage of the *Helicoverpa* life cycle normally lasts about two weeks. However in Autumn and Winter, short daylengths and cool temperatures can trigger a proportion of pupae to go into diapause. This is a dormant phase that allows them to survive in a state of suspended development for several months. When soil temperatures increase in Spring, normal development is resumed and moths emerge soon afterwards.

## Why should over wintering pupae be controlled ?

Diapause generally commences at the end of the cotton season, when the levels of insecticide resistance in *H. armigera* are at their highest. Moths that emerge in the following spring from diapausing pupae are likely to be highly resistant. In fact, these individuals are the major carriers of resistance from one season to the next. Control of over-wintering pupae by cultivation as soon as possible after harvest, is crucial to managing resistance.

## Diapause induction

The proportion of pupae predicted to enter diapause on a given date for several of the

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

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

*Helicoverpa armigera* autumn diapause induction and emergence dates of non-diapausing pupae for major regions (Central Queensland and Macintyre) based on long term average temperatures

Central Queensland

Date of egg lay	Pupation	% Diapause	Non-Diapause emergence
1 February	21 February	0.0	5 March
8 February	25 February	0.0	10 March
15 February	7 March	2.1	19 March
22 February	12 March	15.7	25 March
1 March	22 March	42.0	5 April
8 March	29 March	57.8	15 April
15 March	6 April	74.4	23 April
22 March	15 April	90.9	18 May

Macintyre

Date of egg lay	Pupation	% Diapause	Non-Diapause emergence
1 February	25 February	0.0	11 March
8 February	3 March	0.0	18 March
15 February	11 March	11.4	28 March
22 February	19 March	29.1	6 April
1 March	27 March	46.8	17 April
8 March	5 April	64.4	30 April
15 March	14 April	78.6	20 May
22 March	25 April	90.4	10 June

*Helicoverpa armigera* autumn diapause induction and emergence dates of non-diapausing pupae for major regions (Namoi and Macquarie ) based on long term average temperatures

Namoi

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

Macquarie

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

**Predicted emergence of *Helicoverpa armigera* from diapause in Spring.**

The proportion of diapausing pupae predicted to resume normal development and emerge as moths is given for each of the major cotton growing regions based on long term average temperatures. These predictions were made using the model of diapause termination for *H. armigera* developed by Dr David Murray. Diapause termination is influenced by soil temperatures. The model predictions should be considered as estimates only, because the interac-

tions between soil temperature, moisture, and pupal depth will be strongly influenced by local conditions at the field level. In cool seasons these emergence periods may be delayed by up to two weeks, and in warm seasons emergence will occur earlier than average. Emergence from diapause is a drawn out process that generally takes place over 6 to 8 weeks.

**Spring emergence of *H. armigera* moths from winter diapause**

Region	1% emergence	50% emergence	99% emergence
Central Qld	14-Aug	3-Sep	6-Oct
Macintyre	28-Sep	23-Oct	23-Nov
Gwydir	1-Oct	26-Oct	25-Nov
Namoi	4-Oct	29-Oct	28-Nov
Macquarie	21-Oct	13-Nov	12-Dec
MIA	27-Oct	17-Nov	9-Dec

## Appendix 2

Mensah R, Dillon ML, Khan M, Tann C and Wilson L. 1999. Cotton Insect Pests and their crop hosts. Support Document 3 in Mensah R and Wilson L. (Editors) "*Integrated Pest Management Guidelines for Australian Cotton*", Australian Cotton Cooperative Research Centre, Narrabri. 1999

# Cotton insect pests and their crop hosts

Prepared by Robert Mensah<sup>1</sup>, Martin Dillon<sup>2</sup>, Moazzem Khan<sup>1</sup>, Colin Tann<sup>2</sup> and Lewis Wilson<sup>2</sup>

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

Crop Host \ Pest	Cotton bollworm	Native budworm	Spider mites	Cotton aphid	Green Mirid	Apple dimpling bug	Thrips	Cotton tipworm	Rough bollworm
	<i>Helicoverpa armigera</i>	<i>Helicoverpa punctigera</i>	<i>Tetranychus urticae</i>	<i>Aphis gossypii</i>	<i>Creontiades dilutus</i>	<i>Campylomma liebnechti</i>	<i>Thrips tabaci</i> , <i>Frankliniella schultzei</i>	<i>Crociosema plebejana</i>	<i>Earias huegelli</i>
Adzuki bean	✓	✓	✓		✓				
Barley	✓						✓		
Bean	✓	✓	✓		✓		✓		
Canola	✓	✓	✓	✓			✓		
Chickpea	✓	✓							
Cotton	✓	✓	✓	✓	✓	✓	✓	✓	✓
Cowpea	✓	✓			✓				
Fababean	✓	✓	✓	✓	✓		✓		
Field pea	✓	✓	✓						
Lablab	✓	✓	✓	✓	✓	✓			
Linseed	✓	✓							
Lucerne	✓	✓	✓		✓	✓	✓		
Lupin	✓	✓			✓				
Maize/corn	✓		✓	✓					
Melon	✓	✓	✓	✓					
Mungbean	✓	✓	✓		✓				
Peanut	✓	✓	✓						
Pigeon pea	✓	✓		✓	✓	✓			
Safflower	✓	✓	✓		✓	✓			
Sesame	✓	✓							
Sorghum	✓		✓	✓		✓			
Soybean	✓	✓	✓						
Sunflower	✓	✓	✓	✓	✓	✓			
Tomato	✓	✓	✓						
Wheat	✓			✓			✓		

## Appendix 3

Mensah R, Dillon ML, Khan M, Tann C and Wilson L. 1999. Cotton Insect Pests and their weed hosts. Support Document 4 in Mensah R and Wilson L. (Editors) "*Integrated Pest Management Guidelines for Australian Cotton*", Australian Cotton Cooperative Research Centre, Narrabri. 1999.

# Cotton insect pests and their weed hosts

Prepared by Robert Mensah<sup>1</sup>, Martin Dillon<sup>2</sup>, Moazzem Kahn<sup>1</sup>, Colin Tann<sup>2</sup>, and Lewis Wilson<sup>2</sup>

Most insect pests that attack cotton also utilize one or more weed or native host plants. This table provides a guide to some of the weed and native plants within cotton growing regions that may also support populations of pests. It is not a comprehensive list, and should be taken as a guide only.

Common Name	Pest Scientific name	Cotton bollworm	Native budworm	Spider mites	Cotton aphid	Green Mirid	Apple dimpling bug	Thrips	Cotton tipworm	Rough bollworm
		<i>Helicoverpa armigera</i>	<i>Helicoverpa punctigera</i>	<i>Tetranychus urticae</i>	<i>Aphis gossypii</i>	<i>Creontiades dilutus</i>	<i>Campylomma liebnechti</i>	<i>Thrips tabaci</i> , <i>Frankliniella schultzei</i>	<i>Crociosema plebejana</i>	<i>Earias huegelli</i>
Weed										
Anoda weed	<i>Anoda cristata</i>								✓	
Australian bindweed	<i>Convolvulus erubescens</i>			✓						
Bathurst burr	<i>Xanthium spinosum</i>	✓	✓							
Blackberry nightshade	<i>Solanum nigrum</i>			✓		✓				
Bladder ketmia	<i>Hibiscus trionum</i>	✓		✓						✓
Burr medic	<i>Medicago polymorpha</i>	✓	✓	✓				✓		
Cape honeysuckle	<i>Tecomaria capensis</i>	✓	✓							
Capeweed	<i>Arctotheca calendula</i>			✓						
Cat head	<i>Tribulus terrestris</i>	✓		✓						
Cobblers peg	<i>Bidens pilosa</i>				✓					
Common joyweed	<i>Alternanthera nodiflora</i>					✓				
Clover	<i>Trifolium spp</i>	✓	✓	✓						
Cotton regrowth	<i>Gossypium hirsutum</i>	✓	✓	✓	✓	✓	✓	✓	✓	✓
Crowfoot	<i>Erodium spp.</i>	✓								
Curled dock	<i>Rumex crispus</i>			✓						
Dead nettle	<i>Lamium amplexicaule</i>	✓	✓	✓				✓		
Docks	<i>Rumex spp.</i>			✓	✓					
Flax fleabane	<i>Conyza bonariensis</i>			✓	✓					
Gerbera	<i>Gerbera jamesonii</i>	✓	✓							
Glauca	<i>Haloragis glauca</i>					✓				
Eucalyptus trees	<i>Eucalyptus spp.</i>				✓					
Hairy carpet weed	<i>Glinus lotoides</i>					✓				
Hibiscus	<i>Hibiscus spp.</i>				✓			✓		
Lucerne	<i>Medicago sativa</i>			✓		✓	✓	✓		

Insects

ton insect pests and their weed hosts continued:

Common Name	Pest Scientific name	Cotton bollworm	Native budworm	Spider mites	Cotton aphid	Green Mirid	Apple dimpling bug	Thrips	Cotton tipworm	Rough bollworm
		<i>Helicoverpa armigera</i>	<i>Helicoverpa punctigera</i>	<i>Tetranychus urticae</i>	<i>Aphis gossypii</i>	<i>Creontiades dilutus</i>	<i>Campylomma liebnehti</i>	<i>Thrips tabaci</i> , <i>Frankliniella schultzei</i>	<i>Crociosema plebejana</i>	<i>Earias huegelli</i>
Weed										
Malvastrum	<i>Malvastrum americanum</i>	✓								
Marshmallow	<i>Malva parviflora</i>	✓		✓	✓	✓		✓	✓	
Medic	<i>Medicago spp.</i>	✓	✓	✓	✓					
Melon	<i>Citrullus lantanus</i>	✓	✓	✓						
Mullein	<i>Verbascum virgatum</i>	✓	✓				✓			
Niger seed	<i>Tecomaria capensis</i>	✓	✓			✓				
Noogoora burr	<i>Xanthium spp</i>	✓	✓	✓		✓				
Paper barks	<i>Melaleuca spp.</i>				✓					
Paradoxa grass	<i>Phalaris paradoxa</i>			✓						
Paddy melon	<i>Citrullus colocynthis</i>			✓	✓					
Paterson's curse	<i>Echium plantagineum</i>	✓	✓		✓		✓	✓		
Pigweed	<i>Portulaca oleracea</i>				✓					
Prickly lettuce	<i>Lactuca serriola</i>			✓						
Purple top	<i>Verbena bonariensis</i>				✓					
Saffron thistle	<i>Carthamus lanatus</i>	✓	✓							
Salt bush	<i>Atriplex spp.</i>		✓			✓	✓			
Sida	<i>Sida spp.</i>				✓					
Slender dock	<i>Rumex brownii</i>			✓						
Snapdragon	<i>Antirrhinum majus</i>	✓	✓							
Sow / milk thistle	<i>Sonchus oleraceus</i>	✓	✓	✓				✓		
Trailing verbena	<i>Verbena supina</i>					✓				
Turnip weed	<i>Rapistrum rugosum</i>	✓	✓	✓		✓		✓		
Variegated thistle	<i>Silybum marianum</i>			✓	✓	✓		✓		
Verbines	<i>Psoralea spp.</i>	✓	✓							
Vetch	<i>Vicia spp.</i>	✓	✓							
Weeping willow	<i>Salix babylonica</i>				✓					
Wild sunflowers	<i>Helianthis annuus</i>	✓	✓	✓	✓	✓		✓		
Wild Tobacco	<i>Nicotiana spp</i>	✓	✓							
Wild Turnip	<i>Brassica spp.</i>	✓	✓	✓	✓	✓		✓		
Wireweed	<i>Polygonum arenastrum</i>			✓						

## Appendix 4

Dillon ML. 1999. Guide to planting dates and flowering dates for chickpea trap crops. Support Document 6 in Mensah R and Wilson L. (Editors) "*Integrated Pest Management Guidelines for Australian Cotton*", Australian Cotton Cooperative Research Centre, Narrabri. 1999.

# Guide to planting dates and flowering dates for chickpea trap crops

Prepared by Martin Dillon <sup>1</sup>

This table lists the date at which 50% of plants will flower for a range of planting dates at each of the locations given for the 'Amethyst' chickpea cultivar. It is based on long term average temperatures, and so should be considered as a guide only. Some plants will commence flowering earlier than the dates given, and generally the day on which 5% of plants

are in flower falls about 5 days earlier than the 50% date. The dates at which 95% of plants will be in flower are generally 5 to 10 days after the 50% date. Amethyst chickpea will continue flowering whilst soil moisture is available. Flowering crops are more attractive to *Helicoverpa* than non-flowering crops.

Planting Date	Gunnedah	Narrabri	Walgett	Moree	Goondiwindi
1 <sup>st</sup> June	31-Aug	27-Aug	26-Aug	27-Aug	24-Aug
15 <sup>th</sup> June	10-Sep	7-Sep	6-Sep	7-Sep	4-Sep
1 <sup>st</sup> July	21-Sep	18-Sep	16-Sep	17-Sep	14-Sep
15 <sup>th</sup> July	29-Sep	26-Sep	24-Sep	25-Sep	22-Sep
1 <sup>st</sup> August	8-Oct	5-Oct	3-Oct	3-Oct	2-Oct
15 <sup>th</sup> August	16-Oct	13-Oct	11-Oct	12-Oct	10-Oct
1 <sup>st</sup> Sept	25-Oct	23-Oct	21-Oct	22-Oct	20-Oct
15 <sup>th</sup> Sept	3-Nov	2-Nov	30-Oct	31-Oct	30-Oct
1 <sup>st</sup> Oct	13-Nov	11-Nov	10-Nov	11-Nov	10-Nov
15 <sup>th</sup> Oct	24-Nov	22-Nov	20-Nov	22-Nov	21-Nov
1 <sup>st</sup> Nov	7-Dec	5-Dec	4-Dec	5-Dec	5-Dec
15 <sup>th</sup> Nov	19-Dec	18-Dec	17-Dec	17-Dec	17-Dec
1 <sup>st</sup> Dec	2-Jan	2-Jan	1-Jan	2-Jan	2-Jan
15 <sup>th</sup> Dec	16-Jan	15-Jan	14-Jan	15-Jan	15-Jan

## Appendix 5

Ferguson J, Miles M, Murray DAH, Dillon ML, Kauter G, and Lloyd R. 2000. *Spring Trap Crop Management Guidelines*. Australian Cotton Cooperative Research Centre, Cotton Research and Development Corporation and Grains Research and Development Corporation, 2000.

# Spring Trap Crop Management Guidelines

Compiled by Julie Ferguson<sup>1</sup>, Melina Miles<sup>1</sup>, David Murray<sup>1</sup>, Martin Dillon<sup>2</sup>, Greg Kauter<sup>1</sup>, Richard Lloyd<sup>1</sup> and Richard Sequeira<sup>1</sup>

## Introduction

Trap crops, along with pupae-busting and good in-crop heliothis management are important components of Area-Wide Management (AWM) and Integrated Pest Management (IPM) of heliothis (*Helicoverpa* spp.).

Trap crops could be used at different times during the cropping season.

Here we focus only on spring trap crops used as part of an AWM program.

Trap cropping is still very much in an experimental phase.

Due to the large grower interest in AWM, trials are being carried out throughout most major cotton growing areas from Warren to Emerald.

The Emerald Irrigation Area (EIA) and the Darling Downs are conducting extensive monitored trials with the cooperation of many growers.

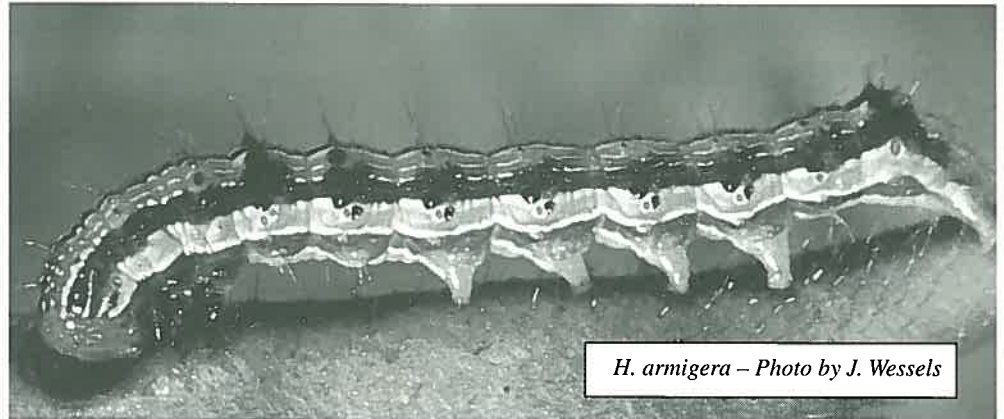
The success of trap cropping is difficult to measure because of the high variability in heliothis populations both within and between seasons.

Indicators of success with AWM may be a delay in the onset of the major population outbreak of *Helicoverpa armigera*, a reduction in the levels of *H. armigera* resistance to chemicals, and an associated reduction in use of chemicals in summer crops.

Information presented in this brochure is the latest information available. However, it is continually evolving, and research into various aspects of trap cropping is ongoing.

## The heliothis problem

Two pest species of heliothis occur in Australia, *Helicoverpa armigera* and *Helicoverpa punctigera*. Both species attack a wide range of crops, but only *H. armigera* is found on monocots (sorghum, maize, cereals). Crops are most attractive in the flowering and grain/fruit and pod fill stage.



*H. armigera* – Photo by J. Wessels

### *H. armigera* breeds locally

Populations of *H. armigera* are thought to be mostly generated locally within our cropping regions. In temperate areas they over-winter as pupae and emerge as moths in spring. In contrast *H. punctigera* is known to breed in inland Australia during favourable winter seasons and migrate to the cropping regions each spring.

*H. armigera* has developed resistance to several insecticide groups and control is becoming less reliable. *H. punctigera* is readily controlled with insecticides. Spring is a potential bottleneck for heliothis populations, with availability of plant hosts influencing the size of the first spring generation.

A spring trap crop is timed to attract mostly *H. armigera* but, depending on the crop used, both species may be attracted.



## Objectives of spring trap cropping

A trap crop, strategically timed to flower in the spring, can help to reduce early season build-up of *H. armigera* on a regional/district scale. This can be approached in different ways:

1. In Central Queensland there is minimal over-wintering of *H. armigera*. Here spring trap crops aim to concentrate local *H. armigera* populations into areas where they can be destroyed.
2. In southern areas, where there is a high incidence of resistant over-wintering *H. armigera*, an area of flowering trap crop acts to concentrate locally emerging *H. armigera* moths during the main emergence period, through October.

The spring trap crop doesn't necessarily aim to draw pressure away from summer crops. *Heliothis* infestations in the trap crop are controlled where necessary and destroyed by cultivation.

## Trap crop selection

An ideal trap crop has the following characteristics:

- highly attractive to *H. armigera*
- nursery for beneficial insects
- low weed potential
- low disease susceptibility
- low potential to host secondary pests and diseases
- relative ease of sowing, establishment and crop management.

Chickpea is a favoured host for heliothis and it can be manipulated (by a late plant) to flower in October. Other winter crops (such as linseed) tend to attract more *H. punctigera* rather than *H. armigera*. *H. armigera* is the main target of trap crops.

Trials have been conducted on the Darling Downs and in Central Queensland to evaluate alternative trap crop options. These results are presented below.

## Alternative trap crop trial results Darling Downs and Central Queensland

### Aims

1. To assess the relative attractiveness of a range of winter crops to *H. armigera*.
2. To determine the potential of these crops as nurseries for beneficial insects.
3. To identify potential alternatives to chickpea for spring trap crops, given the emergence of *Ascochyta* as a major disease of chickpea.

## Darling Downs

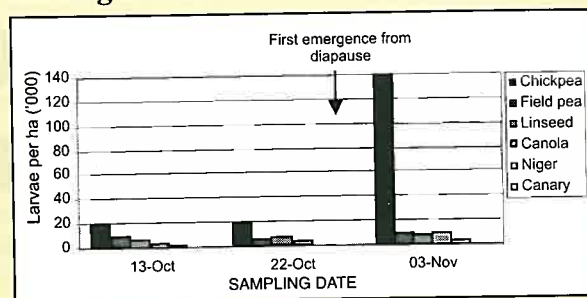


Figure 1. Relative attractiveness of alternative trap crops to *H. armigera* (Brookstead, Qld).

## Central Queensland

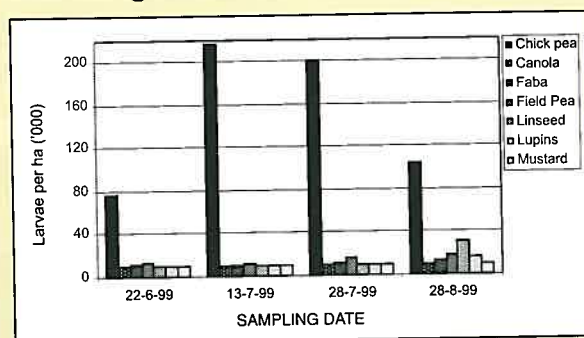


Figure 2. Relative attractiveness of alternative trap crops to *H. armigera* (Emerald, CQ).

## Attractiveness to secondary pests and beneficials

All crops hosted populations of aphids, which attracted generalist predators (eg. lacewings, ladybirds). Parasitism of heliothis by wasp species was high in linseed, field pea and canola (20-30% of larvae). These crops also hosted breeding populations of secondary pests, including green mirid, apple dimpling bug and green vegetable bug. Niger and canary were attractive to a range of predatory bugs and beetles.

## Conclusions

1. Of the crops tested, chickpea is significantly more attractive to heliothis than other spring flowering species. This result reinforces the suitability of chickpea as a spring trap crop, and trials are planned to investigate varietal differences in the attractiveness of chickpea.
2. All crops, other than chickpea, supported diverse populations of beneficial insects. The beneficials are primarily aphid feeders, but many will also feed on heliothis eggs and small larvae.
3. Wheat supported a similar number of aphid-feeding beneficial insects as the non-chickpea crops included in the trial. Potentially, winter cereals are a major source of beneficials, and we need to consider this in the management of these crops eg. chemical choice for the control of armyworm outbreaks in winter cereals.

**Recommendation:** Use chickpea as a spring trap crop.

## FOR MORE INFORMATION CONTACT:

Dr Melina Miles, DPI Farming Systems Institute, Toowoomba  
Dr Richard Sequeira, DPI Farming Systems Institute, Emerald



*Heliiothis larva in chickpea – Photo by J. Ferguson*

## Planting information for chickpea trap crops

### How much should I plant?

Sow 1% of your **cultivated** area or a minimum of 2 ha. This should be planted in rows to allow ease of insect checking. Planting in rows also reduces the disease susceptibility of the crop, by allowing the canopy to dry out.



The optimal area of trap crop required to effectively suppress *H. armigera* on a regional scale is yet to be determined. The relative effectiveness of planting in strips, blocks, or of seed mixes also requires further research.

### Where should I locate my trap crop?

Discuss the positioning of the trap crop with your neighbours to avoid two trap crops side by side. Place the trap crop in a location that is easily accessible for monitoring.



The trap crops that recorded the largest numbers of larvae in 1998, up to 100/m<sup>2</sup>, were grown on 1 metre row spacings, with good seedbed preparation.

### When is the best time to plant?

The recommended planting time is late July to early August – this will give a clear break from the commercial crop in most regions. See Figure 3.

In dryland areas where planting opportunities may be limited due to rainfall, consider delaying flowering of the trap crop by slashing. Regrowth may be slow, or fail, if moisture is limiting.

### Can I slash part of a commercial crop?

An area of a commercial chickpea crop may be manipulated by slashing to delay/prolong flowering until October.

The initial slashing should be done in early to mid September. Slashing prior to podding will delay flowering for approximately 2 weeks.

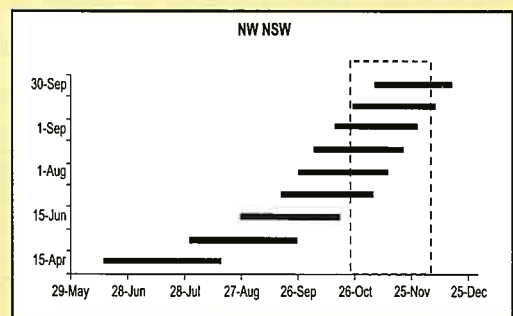
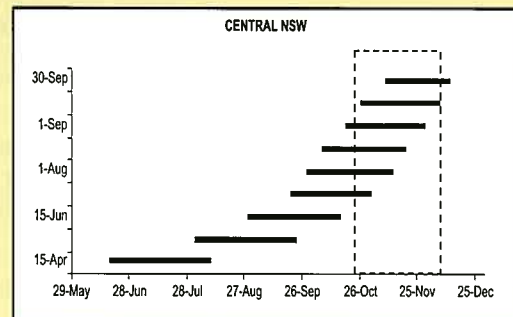
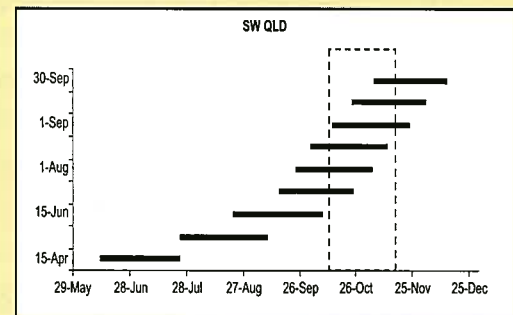
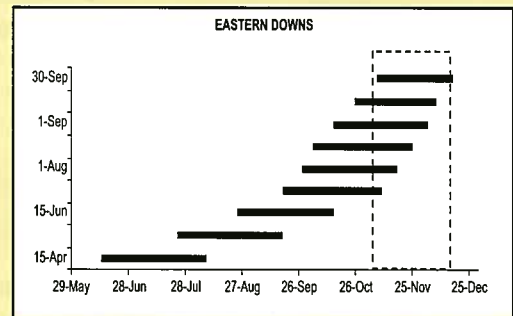
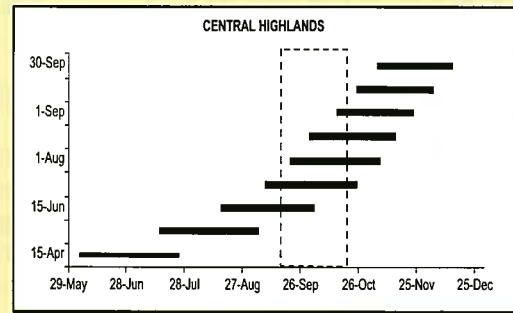


Figure 3. Predicted flowering times of chickpea, planted at approximately monthly intervals Apr–Sep. The period of predicted emergence of *H. armigera* from diapause is indicated by the dashed box. (Chickpea development predictions based on output from the APSIM model).

It may be necessary to slash a commercial crop several times to prolong flowering into October–November to coincide with emergence of *H. armigera* from over-wintering.

Slashing a moisture-stressed crop may result in little, or no regrowth.

### What planting rate should I use?

The recommended plant stand is 20 plants/m<sup>2</sup>. The planting rate does not need to be as high as for a commercial crop because:

- lower plant populations will result in larger plants with lower water requirement and **prolonged flowering**
- the crop will **not** be harvested.

Weed growth may be a problem in low-density stands.

## Agronomy of chickpea trap crops

For information on agronomic advice for chickpea please consult the DPI Farming Systems Institute 'Winter Crop Management Notes' or the NSW Agriculture 'Winter Crop Variety Sowing Guide 2000'.

For specific information please consult your local DPI extension officer or NSW Ag district agronomist.

## Disease Management

Maintaining a healthy, vigorous chickpea crop will ensure the trap crop is most attractive to heliothis. To achieve this requires good disease management.

### Should I treat the seed prior to planting?

Use an inoculant to ensure good, healthy growth of the crop and consider a starter fertiliser.

Treatment for *Botrytis* grey mould, *Phytophthora*, *Pythium* and other soil borne diseases is strongly recommended.

All planting seed should be treated with P-Pickel T<sup>®</sup> for *Ascochyta*.

### What variety is most suitable?

Selecting a variety with disease tolerance needs to be considered.

Table 1. Susceptibility of chickpea varieties to *Ascochyta* and *Phytophthora*.

	<i>Ascochyta</i> Ratings	<i>Phytophthora</i> Ratings
Least Susceptible	Sona, Tyson	Barwon, Jimbour, Norwin
Moderately Susceptible	Jimbour, Heera, Amethyst, Dooen,	Amethyst, Dooen Gully, Sona, Heera
Highly Susceptible	Barwon, Norwin, Gully, Garnet, Bumper	Garnet, Bumper Tyson

Note: All varieties are desi types, except Garnet and Bumper which are kabuli.

## Should I program spray my trap crop with fungicide?

Early application of fungicides is critical in restricting the early development and spread of *Ascochyta*. The following chart indicates the ideal timing for the first two programmed sprays, even if the crop appears free of any disease.

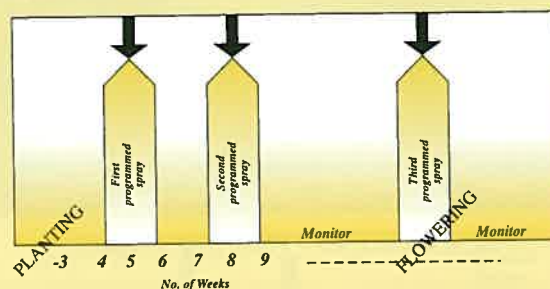


Figure 4. Example of program spray strategy for *Ascochyta* management.

## Management of the trap crop

The ideal trap crop is one that is highly attractive during spring. In southern districts, chickpea trap crops should be flowering during the emergence of over-wintering *H. armigera*. The trap crop must be monitored regularly and destroyed before any pupae emerge as moths.

### When do I scout my trap crop?

All crops should be scouted for insects and disease at regular intervals, particularly to detect early infestations and time destruction. Look for eggs, larvae and moths.

Check at least 5 metres of row at 6 widely spaced sites in the field each time you inspect.

### How do I scout my trap crop?

- **Direct observation:** look at leaves, stems and buds for small white-cream or brown coloured eggs and small larvae.
- **Plant shaking:** to dislodge insect larvae, shake plants onto a white fertiliser bag placed between the rows.
- **Pull flowers apart** to check for small heliothis larvae within the flowers.

### Do I need to spray?

Spraying may be required for two reasons:

- to protect the trap crop from destruction by large infestations of *H. punctigera* if they occur, and to ensure it doesn't become a nursery for *H. punctigera*
- to prolong the usefulness of the trap crop i.e. kill an early infestation of *H. armigera*

To determine whether heliothis present in the trap crop are *H. armigera* or *H. punctigera*, use a LepTon<sup>™</sup> test kit.



Checking of *heliiothis* larvae in chickpea with beatsheet.  
Photo by J Ferguson

Consider using biological products e.g. Gemstar® or Dipel SC®, so there is no pre-selection for resistance, and minimal disturbance to AWM and IPM.

Consider combining Gemstar® or Dipel SC®, with your program fungicide spray if *heliiothis* needs controlling.

### When should I destroy the trap crop?

The ideal destruction date will depend largely on the season. Temperature will influence the timing of *H. armigera* emergence and the rate of development of the larval population. The appropriate timing for plough down is determined by regional monitoring of pheromone traps and crop inspections.

### Why should I destroy the trap crop?

To avoid creating a nursery for *H. armigera*, the trap crop must be destroyed prior to the pupation of the first large *H. armigera* larvae. **If we let any *H. armigera* moths emerge then we are creating a nursery instead of a trap crop.**

### How should I destroy my trap crop?

The trap crop needs to be ploughed out using at least one full-disturbance cultivation. Double discing is the preferred method, however a chisel plough may be used if a disc plough is not available. Slashing the trap crop prior to cultivation will kill some of the larvae and allow easier incorporation of the stubble.

Be aware that where densities are very high, some larvae may survive the slashing and cultivating operation and move into neighbouring crops e.g. seedling summer crops.

Chickpea stubble and regrowth should be buried to minimise the build up of *Ascochyta* in disease confirmed areas and minimise the risk of exposure in disease free areas.

## Risk Assessment

There are many variables that may affect emergence at the local level, such as the amount of stubble cover. Trap crops should be most attractive (flowering) during peak emergence.

The period of predicted emergence from over-wintering is illustrated in Figure 3 (dashed boxes).

Predictions are based on long-term average temperatures. Annual variation can be expected where seasonal temperatures are higher or lower than average.

### How will rainfall affect my trap crop?

Planting opportunity may be limited by rainfall events. To counter this, consider irrigation if available. Rainfall may delay trap crop destruction. If it is close to trap crop destruction time and rain is forecast, consider early destruction to ensure that the trap crop doesn't become a nursery.

In the event that extreme wet weather prevents timely cultivation and there is a risk of the trap crop becoming a nursery, growers may consider applying a pesticide by air. Cultivation should remain an urgent priority as soon as the field dries enough to allow it.

### What if the trap crop runs out of moisture?

If the crop finishes early due to lack of water, or is severely stressed, then it must be ploughed out. The crop will no longer be attractive to *heliiothis*, therefore will have finished attracting moths and will have served its purpose.

### How many is too many?

**One female moth is capable of laying up to 3000 eggs on susceptible crops.**

### What happens if I have *heliiothis* in my trap crop early?

Early infestations of *H. armigera* may require the early destruction of trap crops in some years. If the infestation is *H. punctigera*, then spraying the trap crop may be a more appropriate option.

Using the HEAPS model it is possible to predict the rate of larval development in a crop. Appendix 1 provides a guide to predicted rates of development in a number of regions in New South Wales and Queensland.

### How does a commercial chickpea crop affect the trap crop?

It is important that late planted commercial, regrowth, volunteers or abandoned crops not be neglected and allowed to become nurseries for *H. armigera* and *H. punctigera*. The number and species of *heliiothis* in crops should be monitored, and crops sprayed with an appropriate insecticide if economic threshold levels are reached. Consider desiccation of the crop once main pods are mature. This will make the crop unattractive to any further *heliiothis* infestations and enable earlier harvesting.



Consider using the biological products Gemstar® or Dipel SC® to control *heliiothis*.

## Industry Perspective's

### The Emerald trap-cropping program

Central Queensland (CQ) is now entering its third season of an area-wide IPM program based on the use of trap crops. At the end of the 1998–99 cotton season, the Central Highlands Cotton Growers and Irrigators Association consultative group on research and technical matters was formed. The group meets on an as-needs basis to review the research and extension needs of the cotton industry in the Emerald area.

The objectives of the trap-cropping program are:

1. Minimise the movement of heliothis on to cotton in spring.
2. Minimise migration from cotton to other crops, in and around the irrigation area, at the end of the cotton season.
3. Slow down the build-up of insecticide resistance by eliminating a proportion of the heliothis population in the area in spring and summer.

Emerald growers aim to achieve these objectives by strategic placement of trap crops (chickpea in spring and pigeon pea in summer).

From an area-wide management perspective, the challenges are:

1. The spring population of *H. armigera* is largely determined by the extent of winter dryland cropping in the area. For the most part the *H. armigera* converging on the irrigation area are from a diversity of host plants.
2. In CQ, soil and air temperatures are normally warm enough during the autumn–winter that over-wintering incidence is low. Instead populations over-winter as active breeding populations. Moths generally disperse into other crops before cotton is harvested, and pupae-busting is largely ineffective.

#### 1999 Experience – EMERALD

Chickpea trap crops were planted in early June with an excellent participation rate amongst growers. We estimate that



*Heliothis Moth* – Photo by J. Wessels

75% of Emerald growers planted and fully established their trap crops. The crops picked up large egg lays in early July, and by mid-July were carrying around 30–40 larvae/metre. By early September the trap crops were still carrying in excess of 30 larvae/metre.

After a dry March to August the group met to discuss their options. Two options were decided upon.

**OPTION 1:** Give the crops an additional irrigation in early September to extend maturity to the end of the month, or into early October. This would also mean spraying these crops with a product such as Gemstar® that would not preselect for resistance to conventional cotton chemistry. This action would minimise the number of larvae reaching maturity on the crop as a result of the late July pressure.

**OPTION 2:** Plough out all crops in mid-September to destroy much of the present generation. This would not require an extra irrigation, or crops to be sprayed.

#### The decision

The group decided on the second option for the following reasons:

- It was thought that there was too much risk of the moths escaping out of the system if crops continued longer, creating a problem rather than solving one.
- There was not enough confidence in Gemstar® for it to be recommended on an area-wide basis.
- Not everyone was prepared to give trap crops an extra irrigation.

#### The outcome

Heliothis pressure was very low at the start of the 1999–2000 season with pressure not reaching levels that warranted control until the last week in October. At this stage, cotton crops had been in the ground for as long as 6–7 weeks. There was an increase in moth activity mid-September followed by a sharp decline shortly after the destruction of the trap crops.

Is the light start to the 1999–2000 season a result of the trap cropping program, or was the result inevitable given the lack of significant winter cropping in the area during winter? While the direct impact of the trap cropping program remains unknown, destruction of a large number of trap crops carrying in excess of 30 larvae/metre would have, at the very least, removed a very large number of larvae carrying resistance genes from the previous season.

This has to be of considerable benefit to all summer crops. Therefore, the practice of trap cropping has the potential to go a long way in mitigating the insecticide resistance problem in the area.

The key things to be learned from this season's area-wide IPM experience are:

1. Area-wide IPM plans need to be flexible enough to change as the season changes.
2. Trap crops need to be looked after. There was a strong correlation this season between high insect numbers and the healthier crops.

## Leading researcher, Dr Richard Sequeira, reviews trap cropping

In an attempt to gauge the likely impact of the spring trap crops, egg-laying activity on cotton during the first two months of the season was analysed. Commercial cotton crop scouting data for five farms in the Emerald area over the period 1989–2000, was used in the analysis. The lowest mean egg density for October was recorded in 1997, followed by 1999. However, these two estimates are not statistically different from the average for October in five other years prior to the start of the trap-cropping program. A similar pattern emerges for the month of November. The bottom line is that the direct impact of the trap-cropping program, in terms of a reduction in egg density on cotton early in the season, cannot be distinguished from that of other weather related and external regulatory factors influencing heliothis population dynamics.

A 'favourable' start to the cotton season occurred in several years prior to the introduction of the program. Clearly the current data set may not be adequate for a meaningful assessment of the impact of the trap-cropping program. It is also clear that the winter crop component of the Emerald production system is a crucial factor in determining the pest status of heliothis on late spring and summer crops. Thus, the potential early-season impact of the trap-cropping program on cotton crops in any given year will depend in part on the acreage under rain-fed chickpea, and the degree of overlap with cotton.

The picture emerging from the work so far is that in complex crop production systems, the current form of trap cropping may be of limited usefulness as a stand-alone pest management technique. The importance of winter crop refuges of heliothis and their potential influence on the impact of the Emerald trap-cropping program suggests strongly that trap crops should be used within the framework of area-wide management. Such a strategy would entail development of heliothis population management options for rain-fed winter crops that could be enacted in conjunction with trap crops within irrigation areas.

### The Goondiwindi Experience

Mr John Ferguson, manager of 'Carbucky', Boggabilla NSW, decided to prepare an area of cultivated floodway (previously wheat) for a chickpea trap crop area in February 1999 after viewing local spring trap crops in October the previous year. The consultant, Iain Macpherson, advised the crop be planted on beds as crop establishment may require watering-up west of Goondiwindi.

The aim of the trap crop was to capture the egg-lay from moths emerging from over-wintering in surrounding areas, and help to reduce resistance. It was hoped that the trap crop might also reduce early season insect pressure on the adjacent cotton.

The 35 ha chickpea (variety amethyst) trap crop was planted on 22 July and watered up. Planting rate was 50 kg/ha, resulting in a plant stand of approximately 30 plants per metre, on 1 metre beds. The crop received one inter-row lilliston

cultivation.

The crop grew well from the July planting and commenced flowering about 10 October and reached 100% pods around 5 November.

After heliothis larvae exceeded 1 per metre over 2 checks, the crop was sprayed by ground on 12 October with a mixture of 750 mL Larvin 375® and 1 L Dipel with 1 kg milk powder per hectare.

Pupal sampling under the chickpea during flowering and pod fill indicated that the spray was effective, and that no emergence had occurred from the chickpea area (Table 2).

The trap crop was destroyed on 23 November by Gesner bed renovator with full hill and furrow disturbance followed by Excel Guess Row Averager a week later. Trap crop destruction was delayed by 10 days longer than ideal to accommodate an insecticide trial.

In summary, Iain Macpherson said 'The chickpea were planted so that they would flower as the cotton was emerging. The trap crop was very attractive.

With chickpea it is critical to avoid waterlogging, herbicide damage or cultivator damage. Irrigators should consider raised beds to enhance good drainage but avoid late irrigations. The larvae trapped in the chickpea can only have a positive impact on resistance management and may possibly reduce early season sprays.'

Table 2. Result of pupae sampling under the chickpea trap crop, 1999. 'Carbucky', Boggabilla.

Date	Number/ha
21/10/99	0
27/10/99	2000
1/11/99	0
11/11/99	10 000

### Trap Cropping: an Extension Officer's Perspective

As a member of the Gwydir Valley cotton group, James Quinn, Cotton CRC Industry Development Officer from Moree, reports on their progress.

In the winter of 1999, after a summer which would rather be forgotten in terms of insect control in the Gwydir Valley, a group of cotton growers decided to investigate alternate management practices to assist in the control of heliothis pests.

This group visited the area-wide management project on the Darling Downs in Qld, and decided that this was an interesting approach worth investigating in the Gwydir Valley.

There were several aims of the chickpea trap cropping program:

1. To prove it could be done in this area.
2. To establish an attractive crop without affecting the local pulse industry and not creating a nursery for *Ascochyta* blight.



Goondiwindi Group – Photo by G. Kauter

3. To monitor trap crops to determine their effectiveness in managing *Heliothis* populations.

After the first season of the program the aims of chickpea trap cropping have been achieved. *Heliothis* populations were not significantly reduced on an area-wide basis. This may be achieved when more growers adopt this technique.

Two systems were trialed in the Gwydir Valley:

1. Planting in the recommended commercial chickpea planting window, and controlling maturity through slashing.
2. Planting in the first week of August and flowering in October–November.

Crops planted early and slashed were not as attractive, were more difficult to manage and had greater disease infection than later planted crops. On average, there were 2 larvae/m in early planted chickpea and 9 larvae/m in later planted chickpea. The highest population was 17 larvae/m. The predominant species trapped was *H. armigera* which was very encouraging. Cooperation amongst cotton growers was excellent, and all growers are to continue the trap cropping program next season.

In the coming year I will be promoting chickpea trap crops as a management option for *H. armigera*. Experience of the first year gives me a lot of confidence that improvements can be made to the system in the coming years. Encouraging more growers to undertake this program is seen as a hurdle to overcome, given that results are not overly tangible in the short term.

### Assessment of chickpea, field pea and faba bean for suitability as dryland spring trap crops for *Helicoverpa* spp. in the Macquarie Valley.

In the 1999–2000 season a large plot trial (10 ha/crop) was undertaken to assess the suitability of faba bean, chickpea and field pea as spring trap crops for *Helicoverpa* spp. in the Macquarie Valley. Amanda Mills, agronomist, and Matt Secombe, farm manager, report on the results of the trial at ‘Wambandry’, Warren, NSW. The results from sweepnet sampling are summarised in Figure 5.

Sampling on 29 October prior to crop destruction showed a

total of 7, 35 and 116 larvae and pupae per 3 metres in faba bean, chickpea and field pea, respectively. Input costs for the 3 crops are presented in Table 3.

Table 3. Comparison of input costs for alternative spring trap crops ‘Wambandry’, Warren.

Input	Chickpea	Faba bean	Field pea
Seed	64	40	24
Inoculation	0.5	0.5	0.5
Herbicide			
(3 L Stomp/ha)	27	27	27
<b>Total Cost \$/ha</b>	<b>91.5</b>	<b>67.5</b>	<b>51.5</b>

### Discussion:

It was apparent from the results that sweep netting grossly underestimated the number of larvae in the field pea crop due to the density of the crop. However, it appeared adequate for the faba bean and chickpea. Sweep netting allowed mirid and predator numbers to be assessed.

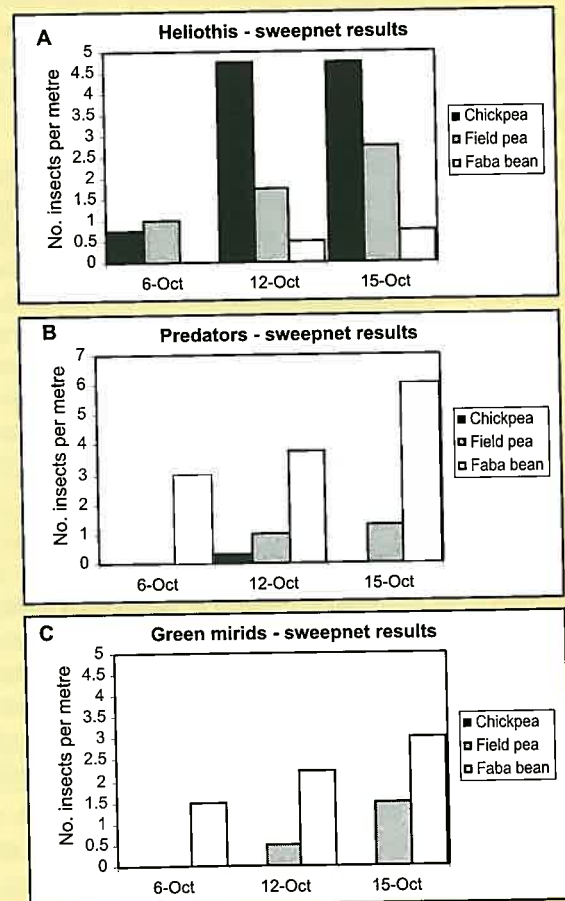


Figure 5. Abundance of *Heliothis*, predators and green mirids in alternative spring trap crops. ‘Wambandry’, Warren. 1999–2000.

Mirid numbers were high (Figure 5C) and the option of spraying these was considered. Temik® (3 kg/ha) should have been working during this early stage of cotton growth (15/10/99) in the adjacent fields. Although it was not necessary to control mirids in the trap crop, this may be something to monitor in the future.

In terms of agronomic factors, production cost and attractiveness to *H. armigera*, field pea is best suited to our dryland spring trap crop scenario.

#### **Future direction of spring trap cropping in the Macquarie Valley:**

- field pea are the cheapest to grow and the most tolerant of poor drainage areas
- good predator numbers in field pea/faba bean
- look at the use of Gemstar® in field pea (to extend life of trap crop)
- greater monitoring of mite and aphid populations in these crops to determine if they may become a source of these pests that later move into adjacent cotton crops
- timing of planting (17 July, field pea) appeared ideal for our situation
- trap crops require as a minimum a double discing to gain maximum control of heliothis in these crops at destruction (especially the very dense field pea).

#### **From the farmer's mouth**

Peter Thomas, spokesperson for the Brookstead pilot study area answers a few questions on trap cropping.

*What are your aims and expectations with regard to the trap crop?*

Mainly we aim to decrease the first spring generation of heliothis and subsequently reduce the numbers in cotton. My expectation is that if enough people plant trap crops we will make inroads into the whole heliothis population.

*How did you think the trap crop performed?*

We have been very satisfied so far. In 1998 we had high numbers in our trap crops, up to 60 larvae/m. In 1999 there weren't the numbers, but they were still there, which is achieving the aim of getting moths to lay eggs in the trap crop rather than somewhere else. I expect that could only be doing good.

*Were you happy with the number of larvae in the trap crops?*

We were very impressed in 1998. In 1999 numbers were lower, but then we didn't have the pressure across the whole region either. Even if we are reducing the population by 10% it is all helping.

*What have been the main differences in the two years that you have planted trap crops?*

Seasonally 1998 was a very wet year; 1999 was a very dry year, however we didn't water the crop. The trap crop in 1998 was a very well growing succulent crop with high larvae numbers. Given that we have had two very different years, both trap crops seem to have done the job. We will be

planting a trap crop again this year.

*What do you think are the benefits?*

We are killing a proportion of the first generation – we aren't killing every heliothis, but it all helps. It has also made everyone start thinking about tackling the heliothis problem from a different angle. People are thinking laterally and everyone is thinking! Trap cropping incorporates a much wider approach to heliothis control. Commercial chickpea growers have been able to see that large areas of uncontrolled crop, or re-growth, can become a huge nursery for heliothis. This is resulting in closer monitoring and spraying of commercial crop. This effectively creates larger areas of trap crop, not nursery crops. One of the main benefits is that we aren't just relying on chemicals to control this pest.

*Any practical hints for growers who may be new to trap cropping?*

We put a lot of our trap crop into odd corners of paddocks, areas that were a problem for cotton machinery. We maintained the trap crops as if it was a commercial crop. It wasn't just planted and left. Many people in our area had joint plantings with neighbours, one farmer went around and planted a number of farms to be time efficient. Most of the trap crops around Brookstead were planted on stored moisture and rainfall. Not having to pre-irrigate is the best plan I think, that way it doesn't become an extra task that may not get done. Trap crops are not a major imposition to plant, especially with co-operation between a few neighbours.

*What impact has trap cropping had on increased/decreased communication with your neighbours?*

There has been a great deal of co-operation with neighbours to decrease the workload in relation to trap crops. The whole program (not just trap crops) has been of tremendous benefit. Regular, informal get togethers are valuable. This trial program has been a great benefit in increasing communication between farmers. Most farmers in the area are embracing the different techniques in the trial, of which trap cropping is one.

*What impact has it had on IPM?*

We are part of a large intensive cropping region and have found it difficult to keep IPM going on our farm beyond December. This is largely due to the impact of other spraying in the region and the constant onslaught of heliothis pressure. The first step that we want to achieve is to keep our IPM program going into January. In order to achieve this we need to have an area-wide management approach, of which trap cropping is an important part.

*What do you think are the potential benefits for IPM?*

It is an important tool for an IPM program. Trap cropping has made us aware that there is more than one way to kill a heliothis. We can extend the trap cropping idea into other areas of the farming system, such as the use of Gemstar® on grain sorghum crops. This trial program is getting everyone thinking. We are not just relying on chemicals as we have done in the past; which aren't working anyway. In the future I'd like to see trap cropping being practiced in all cropping areas.

The following recommendations are taken from the 'Chickpea 2000—Management strategies for the Northern Region', produced by DPI Farming Systems Institute and NSW Agriculture. For more information please consult this brochure. For additional information call your local DPI extension officer or NSW Ag district agronomist.

**Eastern Zone** (to the east of Moonie and Goondiwindi in QLD; to the east of the Newell Hwy in NSW)



**All chickpea stubble paddocks should be regarded as now carrying a high level of disease infection capable of infecting subsequent crops.**

Most chickpea crops in this zone are expected to contain some level of *Ascochyta* and could result in serious economic losses if not effectively controlled.

- On-farm hygiene (fallow management)

The *Ascochyta* fungus can only carry over from one season to the next on infected seed, chickpea stubble, and volunteer chickpea plants. It can only survive on chickpea stubble, and once buried by cultivation will only survive for 2–4 months (normally the length of time it takes for the stubble to decompose).

- Paddock selection

Growers should aim to separate this year's chickpea crop from last year's chickpea stubble by a minimum distance of 500 m to 1 km.

- Avoid susceptible chickpea varieties
- Plant quality low risk seed

All seed must be properly treated with P-Pickel T<sup>®</sup> and obtained from a source where *Ascochyta* was not detected (low-risk seed).

- Monitor the crop closely

After the first two program sprays the crop should be monitored every 2 weeks, or 7–10 days after a rainfall event or heavy dew. If *Ascochyta* is detected then apply the fungicide prior to further rain.

- Modify application equipment for fungicide use

Fungicides used for control of *Ascochyta* are protectants and need thorough coverage over the entire plant to be effective.

- Chickpea rotation

Maintain a rotation of at least three years between chickpea crops to minimise the risk of *Phytophthora* and *Ascochyta* carryover.

**Western Zone** (to the west of Moonie and Goondiwindi, not including St George and Roma in QLD; to the west of the Newell Hwy in NSW)



**Rapid build-up of *Ascochyta* infection in chickpea stubble has occurred over the summer months, and most chickpea stubble paddocks should be regarded as carrying a significant level of disease.**

*Ascochyta* is less of a problem in the western zone, but precautions need to be taken.

- On-farm hygiene

Adopt the strategy as outlined for the eastern zone.

- Obtain low-risk seed

Preferably from within your own area.

- Treat with P-Pickel T<sup>®</sup>
- Apply one programmed spray and monitor

One fungicide spray should be applied 4–6 weeks after planting, and then closely monitor the crop.

**Maranoa–Balonne Zone** (including Roma, St George and Dirranbandi)

While *Ascochyta* was undetectable throughout this region during the 1999 growing season, significant levels of disease have been identified in chickpea stubble paddocks during the summer.

Growers in this region are advised to either adopt the western zone strategy of at least one program spray, or at the very least to have an agronomist intensively monitor the crop from 4–6 weeks after planting.

**CQ–Burnett Zone** (including Emerald, Biloela, Theodore, Byee and Kingaroy)

No *Ascochyta* has currently been detected in this region. Growers and seed handlers are encouraging a voluntary system whereby all parties avoid bringing in chickpea (an *Ascochyta* infection) from outside this region.

All planting seed should be sourced from within this region and treated with P-Pickel T<sup>®</sup>.

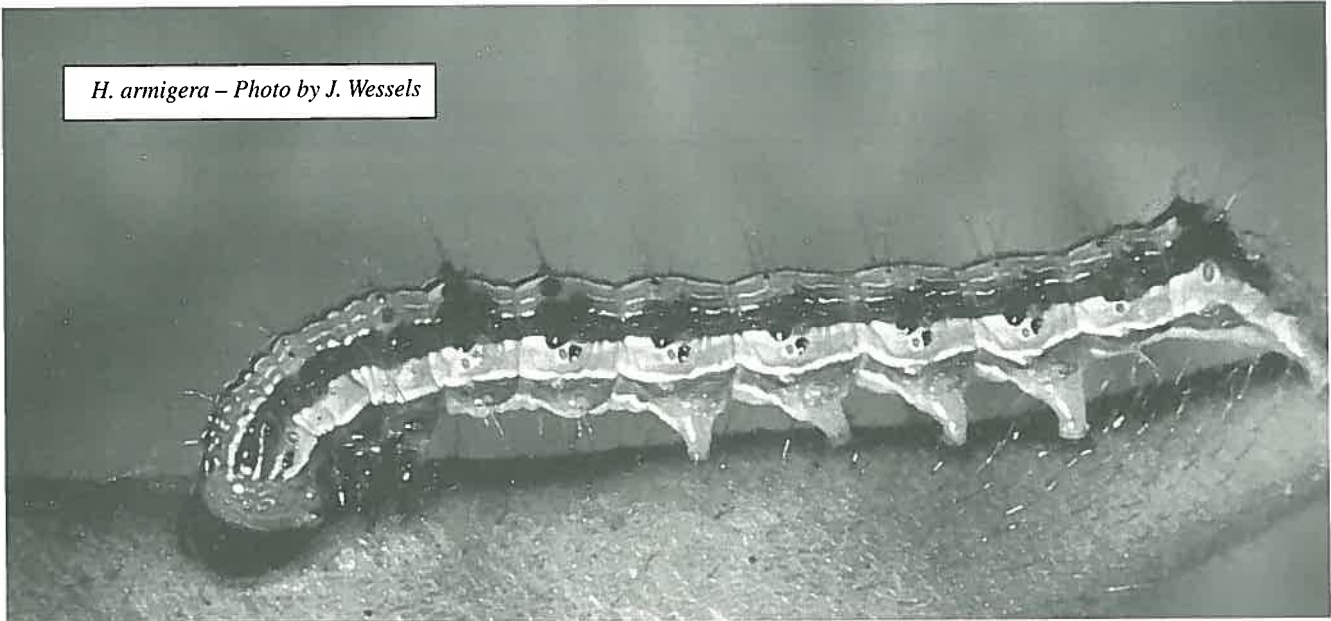
**APPENDIX 1. Predicted heliothis development times based on HEAPS model simulations using long term average daily temperatures. Note that insects are presumed to begin at half-way through the initial stage on the starting date. Also note that in any given period temperatures may vary substantially from the long term averages, and therefore this table should be used as a guide only.**

Starting date and stage		Central NSW Predicted days until...			North West NSW Predicted days until...			South West Qld Predicted days until...			Darling Downs Qld Predicted days until...			Central Highland Qld Predicted days until...		
		Large	Pupa	Moth	Large	Pupa	Moth	Large	Pupa	Moth	Large	Pupa	Moth	Large	Pupa	Moth
9 Sept	W. egg	34	53	78	34	52	74	29	45	64	32	50	70	22	35	51
	Small	13	38	64	12	35	62	12	30	52	12	34	59	9	24	42
	Medium	5	28	56	5	27	55	5	24	48	5	26	54	4	19	38
	Large	-	14	46	-	13	41	-	13	40	-	13	41	-	10	30
16 Sept	W. egg	32	50	75	32	49	69	27	42	60	30	47	66	20	31	47
	Small	12	34	61	12	34	59	10	28	48	12	32	56	9	22	39
	Medium	5	27	54	5	25	53	5	21	44	5	25	51	4	17	35
	Large	-	13	43	-	13	41	-	11	37	-	11	39	-	9	28
23 Sept	W. egg	30	47	70	30	47	66	24	38	56	27	44	63	19	31	46
	Small	12	33	59	11	32	56	9	26	46	11	29	51	7	20	36
	Medium	5	25	52	5	25	51	4	20	41	5	23	47	3	15	32
	Large	-	13	41	-	11	39	-	10	34	-	11	39	-	8	26
30 Sept	W. egg	29	47	64	28	43	62	22	35	52	27	42	61	18	29	43
	Small	10	30	55	12	31	53	9	25	43	9	27	47	7	18	34
	Medium	5	24	51	5	24	48	4	19	39	5	21	44	3	15	31
	Large	-	10	38	-	10	38	-	9	31	-	10	36	-	7	24
7 Oct	W. egg	27	44	63	26	42	60	21	33	49	25	39	58	16	27	41
	Small	11	29	54	10	27	48	9	23	41	9	26	45	6	18	33
	Medium	5	23	48	4	22	44	4	18	36	4	20	40	3	14	29
	Large	-	11	38	-	10	36	-	9	29	-	10	34	-	7	23
14 Oct	W. egg	26	41	59	24	39	57	19	31	46	23	37	56	16	26	40
	Small	9	26	49	10	27	46	8	21	38	9	25	44	6	17	31
	Medium	4	20	44	4	20	41	3	16	34	4	19	38	3	14	28
	Large	-	10	37	-	10	34	-	8	27	-	9	31	-	7	22
21 Oct	W. egg	24	39	55	23	36	53	19	30	44	20	33	52	15	25	39
	Small	9	27	46	9	25	43	7	19	35	9	23	42	6	16	30
	Medium	4	19	42	4	19	38	3	15	32	4	18	37	3	13	27
	Large	-	9	34	-	9	31	-	8	26	-	9	28	-	6	21
28 Oct	W. egg	23	37	54	20	33	49	17	28	42	19	32	49	15	25	38
	Small	9	25	44	8	23	41	6	18	33	7	21	40	6	16	30
	Medium	4	20	39	3	18	36	3	14	30	4	16	35	3	12	26
	Large	-	9	34	-	7	28	-	7	24	-	8	27	-	6	20
4 Nov	W. egg	22	35	50	19	31	46	17	27	41	19	31	47	15	25	37
	Small	9	25	41	8	21	38	7	18	32	7	20	39	6	16	29
	Medium	3	18	37	3	17	35	3	14	28	3	16	35	2	12	26
	Large	-	10	30	-	8	27	-	7	23	-	8	27	-	6	20
11 Nov	W. egg	21	33	49	18	30	44	15	25	38	19	31	45	15	24	36
	Small	9	23	40	7	20	36	6	17	31	7	20	37	6	16	28
	Medium	3	18	34	3	15	32	3	13	27	3	15	33	2	12	25
	Large	-	9	28	-	8	26	-	7	21	-	8	27	-	6	20

## More Information

- Australian Cotton CRC website. <http://www.mv.pi.csiro.au>
- 'Chickpea 2000—Management strategies for the Northern Region', produced by DPI Farming Systems Institute and NSW Agriculture.
- CropLink Pest Updates  
'Know the enemy: *Heliothis ecology and biology for better control*' by J Boddington & D Murray DPI/FSI.  
'Chemical-free *heliiothis pupae control*' by J Boddington & D Murray DPI/FSI.  
CropLink information series brochures available by phoning 13 25 23.
- DPI Farming Systems Institute 'Winter Crop Management Notes'
- *EntoPak*, available from the Technical Resource Centre at the Australian Cotton Research Institute.
- *Integrated Pest Management Guidelines for Australian Cotton* by Robert Mensah and Lewis Wilson.
- Winter Crop Variety Sowing Guide 2000. F.J. McRae NSW Agriculture.

*H. armigera* – Photo by J. Wessels



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### DISCLAIMER

*Information contained in this publication is provided as general advice only. For application to specific circumstances, professional advice should be sought. The Department of Primary Industries, Queensland, and the Australian Cotton CRC have taken all reasonable steps to ensure the information contained in this publication is accurate at the time of publication. Readers should ensure that they make appropriate enquiries to determine whether new information is available on the particular subject matter.*



CSIRO<sup>2</sup> – NSW Agriculture – Department of Primary Industries Queensland<sup>1</sup> – NT Department of Primary Industry and Fisheries – Agriculture Western Australia – University of New England – University of Sydney – Cotton Research & Development Corporation – Cotton Seed Distributors – Queensland Cotton – Western Agricultural Industries – Twynam Cotton

## Appendix 6

Wilson LJ, Mensah R, Dillon M, Wade M, Scholz B, Murray DA, Heimoana V and Lloyd R. 2002. Impact of insecticides and miticides on predators in cotton: October 2002 update. Support Document 1 in: Mensah, R. and Wilson, LJ. (Editors): *Integrated Pest Management Guidelines for Australian Cotton*. Australian Cotton Cooperative Research Centre, Narrabri, NSW.

<http://cotton.pi.csiro.au/Publicat/Pest/>



**Table 2 : Impact of insecticides applied at planting or as seed treatments on key beneficial groups in cotton (high = more disruptive)**

Insecticides	Rate (g ai / ha)	Main Target Pest(s)					Persistence <sup>6</sup>	Overall <sup>7</sup>	Beneficial group				
		WW	Mite	Mir.	Aph.	Th. <sup>5</sup>			Predatory beetles <sup>1</sup>	Predatory bugs <sup>2</sup>	Spiders	Wasps & Ants	Thrips
<b>At Planting</b>													
Aldicarb	450		✓	✓	✓	✓	medium -long	very low <sup>3</sup>	v low	v low	v low	v low	v high
Phorate	600	✓	✓	✓	✓	✓	medium -long	very low <sup>3,4</sup>	No data	No data	No data	No data	v high
Carbosulfan	750-1000	✓		✓		✓	medium -long	very low <sup>3,4</sup>	No data	No data	No data	No data	v high
Chlorpyrifos	250-750	✓					medium	very low <sup>4</sup>	No data	No data	No data	No data	No data
<b>Seed Treatments</b>													
Thiodicarb	500 g ai / 100kg seed					✓	short	very low <sup>3</sup>	v low	v low	v low	v low	high
Thiodicarb + Fipronil	259 + 12 g ai / 100kg seed	✓				✓	short-medium	very low <sup>3,4</sup>	No data	No data	No data	No data	high
Imidocloprid	525 g ai / 100kg seed	✓			✓	✓	medium	very low <sup>3</sup>	v low	v low	v low	v low	v high
Imidocloprid	700 g ai / 100kg seed	✓			✓	✓	medium	very low <sup>3,4</sup>	No data	No data	No data	No data	v high
Thiomethoxam	280 g ai / 100kg seed	✓			✓	✓	medium	very low <sup>3,4</sup>	No data	No data	No data	No data	v high

1. Predatory beetles – ladybeetles, red and blue beetles, other predatory beetles
2. Predatory bugs – Big-eyed bugs, minute pirate bugs, brown smudge bugs, glossy shield bug, predatory shield bug, damsel bug, assassin bug, apple dimpling bug
3. Except for effects on thrips which are predators of mites. Note that aldicarb and phorate will also control mites.
4. Based on observations with other soil or seed applied insecticides.
5. WW., wireworm; Mir, mirids; aph, aphids; th., thrips
6. Persistence; short, 2- 3 weeks; medium, 3-4weeks, long, 4-6 weeks

7. Impact rating (% reduction in beneficials following application); very low, less than 10%; low, 10-20%; moderate, 20-40%; high, 40-60%; very high, > 60%

**DISCLAIMER** Information provided is based on the current best information available from research data. Users of these products should check the label for further details of rate, pest spectrum, safe handling and application. Further information on the products can be obtained from the manufacturer.

### Important Use of Pesticides

Pesticides must only be used for the purpose for which they are registered and must not be used in

any other situation or in any manner contrary to the directions on the label.

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

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

Pesticides may contaminate the environment. When spraying, care must be taken to avoid spray drift on to adjoining land or waterways. Residues may accumulate in animals fed any crop product, including crop residues, which have been sprayed with pesticides. In the absence of any specified grazing withholding period(s), grazing of any treated crop is at the owner's risk. A good way to achieve this is to develop a Spray & Drift Management Plan and also become a BMP accredited grower.

## Appendix 7

Hickman M, Larsen D and Dillon ML. 2002. *Guidelines for assessing pupae risk for dryland cotton growers*. Cotton Information Sheet July 2002. Australian Cotton Cooperative Research Centre, Narrabri.



# COTTON information sheet

## Guidelines for Assessing Pupae Risk for Dryland Cotton Growers

May 2002

Mark Hickman<sup>1</sup>, David Larsen<sup>1</sup>, Martin Dillon<sup>2</sup>, Entomologists & the Farming System Extension Team from the Australian Cotton CRC

### Introduction

This document aims to assist dryland growers in the difficult decision of whether or not to till for pupal control. A tillage operation can carry a conflict between soil related issues (moisture retention, erosion control, compaction) and non chemical insect management. Heliothis control by pupal disruption lowers the threat of resistance as well as lowering potential insect pressure on farm in the following season.

### Key Points

- Information contained in this document is NOT Relevant for Ingard® crops, as fields containing these varieties must be tilled following harvest to remove potentially Ingard® resistant Heliothis pupae. This is a legal requirement of growing the crop.
- There is a great variation in the effectiveness of different types of tillage for killing pupae, depending on the implement and soil conditions at the time. If a grower conducts effective tillage then this negates the need to sample for pupae (see CRC Information Sheet "Has Your Pupae Busting Been Effective May 2002)
- Many dryland crops do not produce diapausing Heliothis pupae as the crops mature well before

diapause starts. Crops should be sampled for larvae at defoliation. This may save time and effort doing more difficult sampling for pupae later in the season and may identify problem fields

- Pupa sampling will enable growers to determine which fields have high numbers of Helicoverpa pupae. Allowing fields to be prioritised for tillage
- Tillage and the associated operational costs can be avoided when sample numbers are below the guideline requirements. A small effort in sampling could save many dollars



Figure 1 Heliothis pupae . Note that when stimulated the tail of live pupae will wiggle from side to side. Generally, pupae which have been parasitised show little tail movement. However, this is highly dependent on the type of parasitoid involved,

- Pupal control should occur if pupal numbers are above 1 pupae in 10 metres after the Regional Diapause Starting Date. Refer to the flow chart on page 2 for the decision process.

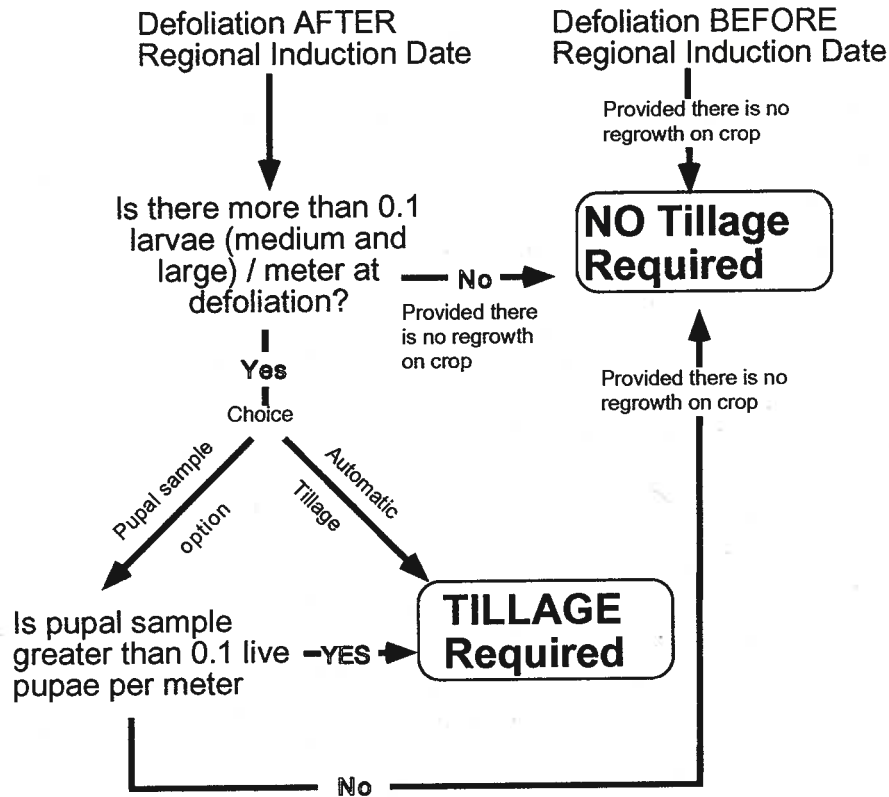
### Information required for the decision process

1. Dates for Heliothis diapause induction in your region
2. Date of field assessment of larvae or pupae
3. Insect pressure at the time of the first defoliation date
4. Pupal numbers derived from soil sampling after harvest
5. Level of cotton regrowth and associated Heliothis activity.



## Do I need to Till or Sample ?

What is my crop defoliation date?



### The Importance of pupae control in all crops

Over-wintering (diapausing) pupae have the potential to create a problem for you and your neighbours in the following season. Larvae remaining in the crop at the end of the season have normally been exposed to some level of chemical control and therefore have been selected for resistance by 1 or more chemical groups.

There is an overwhelming likelihood that the pupae that go into diapause at the end of the season will be *H. armigera*. Insecticide resistance has been a major problem in this species. Diapausing populations which survive the winter will initiate populations in your local area again in the spring. The moths that emerge from these pupae are the primary carriers of resistance from one season to the next.

To put numbers in perspective 1 pupae per metre is equivalent to 10000 pupae per hectare. If we assume a mortality figure of 50% this still leads to on average 5000 emerging moths per

hectare. Half of these will be females that have the potential to lay 1500 eggs each - the result - a lot of eggs!

Pupae control by tillage is the primary way of controlling resistant insects without adding to resistance selection.

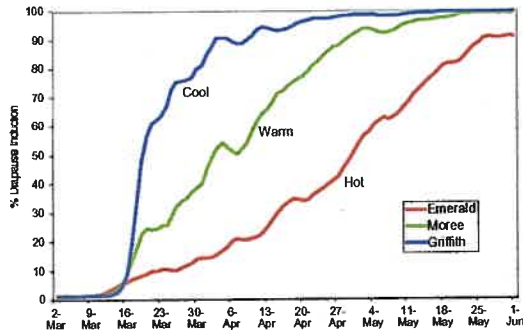
### Diapause - a handy physiological feature allowing *Heliothis* populations to endure !!

The pupal stage of the *Helicoverpa* life cycle normally lasts for about 2 weeks however in autumn and winter a combination of short day lengths and cool temperatures can trigger a proportion of the pupae to go into diapause. This is a dormant phase that allows them to survive in a state of suspended development for several months. Most diapausing pupae will not emerge as moths until October of the following spring.

Those pupae which don't enter diapause will continue to develop during autumn and will emerge in late autumn. In most regions these are unlikely

to survive, but as we move further north, then the non diapausing component increases and these moths will continue to breed during the winter in tropical areas.

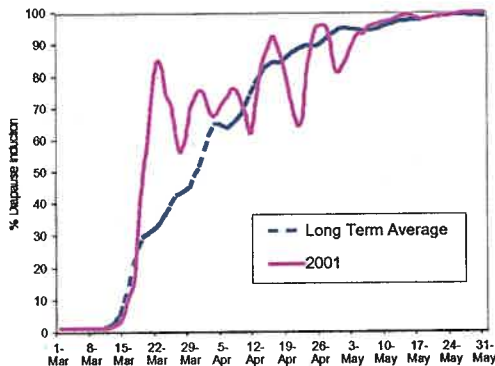
Comparison of % Diapause Induction in cool, warm and hot regions



Graph 1 Helicoverpa Autumn diapause induction based on long term average temperatures (From Martin Dillon CSIRO)

When soil temperatures increase in spring diapausing pupae resume normal development and moths emerge from the soil soon afterwards. The rate at which larvae enter diapause varies from region to region. This rate is determined by temperature and day-length. The further north you travel, then the shorter the diapause period becomes. This is primarily due to higher average temperatures mid year. This is why pupal induction graphs in this document vary in the commencement and duration of diapause.

Boggabri % Diapause Induction



Graph 2 variation in numbers entering diapause can be large within a season due to climatic influence

**Conditions leading to high risk of diapausing pupae following a crop that may require tillage.**

- Regrowth
- High late season pressure

- Management factors promoting late maturity including:

Late sow  
High N  
Late rain

**Conditions leading to low risk of diapausing pupae following a crop.**

- Low stage 3 season insect pressure
- Early crop that matures and is defoliated before the diapause induction date for pupae
- No Regrowth / Alternative hosts
- Active biological control processes including:

Predators (insects, mice)  
Parasites  
Disease / Gemstar

After the induction date is reached the proportion of pupae entering diapause rapidly increases (Graph1).

The actual proportion of Helicoverpa entering diapause will vary with the season Graph 2 shows the predicted proportion entering diapause for one season in one region compared to the mean

**When regrowth occurs:**

Regrowth following crop defoliation can be a haven for Helicoverpa that is often overlooked. Regrowth can allow another generation of larvae to survive and pupate in the field - a generation which will usually not be noticed, but which can enter diapause and survive the winter. If regrowth occurs, sample for medium or large larvae in the regrowth or conduct another pupal survey to ensure that reinfestation has not occurred.

**How do I sample for pupae within fields and what is the critical value?**

To assess the level of pupae in the field an assessor is required to dig a minimum of 10 well-spaced meters. Do not sample near field edges, move at least 40-50 metres into the field for your first sample. The more spaced the sample points the better the indication you have about what is happening. This sampling approach allows for within-field variation. (The critical level is 1 live pupae in the 10 metres sampled).

Select either 1 linear metre in a hill configuration or 1 square metre on a flat configuration:

- In gradual stages, carefully scrape away the top few centimetres to expose any emergence holes.
- Emergence holes are about the diameter of a lead

pencil or an adult small finger.

- If a hole has been located it is necessary to dig down to the chamber containing the protected pupae.
- It is rare to find the pupae deeper than 12 cm.



Photo 2 Scraping surface to reveal tunnels

- After the pupal chamber is located it will contain either a live pupae Photo3 (often active- wiggling tail when touched), or parasitised (non-active) or the remains of a pupae (moth emerged or parasitised). See Australian Cotton CRC Information sheet "Has your Pupae busting been Effective"
- Only count live pupae.



Photo 3. Pupae exposed at the end of pupal chamber

Most pupae are concentrated under the plant canopy, although pupae can be found throughout the bare skip areas in dryland cotton. If you are considering zonal tillage on a flat system you will need to sample between the plant rows to ensure pupae will be disturbed by the equipment you plan to use.

**When do I need to till by?**

The diapause state is broken by environmental conditions at different times in different regions. In general the warmer the region, the shorter the

time the pupae spends in the diapause state. Tillage should be done as soon as possible after the harvesting of the cotton crop, but must be completed before the commencement of crop emergence. Early cultivation will expose the pupae that are disturbed but not killed to the rigours of winter, rainfall, and predators. The following table outlines the emergence percentage using average regional temperatures

Region	% emergence		
	1%	50%	99%
Central Qld	14-Aug	3-Sep	6-Oct
Macintyre	28-Sep	23-Oct	23-Nov
Gwydir	1-Oct	26-Oct	25-Nov
Namoi	4-Oct	29-Oct	28-Nov
Macquarie	21-Oct	13-Nov	12-Dec
MIA	27-Oct	17-Nov	9-Dec

Spring emergence of *H.armigera* moths from winter diapause (data from Martin Dillon CSIRO)

The necessity for controlling pupae may be lowered by :

- Hot regional climate. In hot northern regions (Central Queensland) larvae are unlikely to diapause
- Avoiding over fertilisation with nitrogen (to avoid late season vegetative growth)
- Choosing the correct variety for the sowing date.
- Using insecticide chemistry that minimises effects on beneficial especially hymenoptera (parasitic wasps) that can play a large role in controlling late season larvae and pupae.
- Applying good farm hygiene - avoiding crop hosts and weeds that support *Heliothis*.
- Applying good integrated pest management (See the Australian Cotton CRC IPM Guidelines)
- The use of late season trap crops.
- Removing cotton plants or controlling regrowth after harvest.

**Acknowledgments**

Gary Fitt CSIRO, Dave Murray QDPI, Lewis Wilson CSIRO, Robert Mensah NSW Ag, Colin Tann CSIRO, Julie Ferguson QDPI, Murray Schoenfisch USQ, Cheryl Mares CSIRO, Australian Cotton CRC Farming Systems Extension Team.

## Appendix 8

Dillon ML 1999. Trap cropping attractiveness and performance. Invited presentation. *Proceedings of the Cotton Production Seminar* (Cotton Consultants Australia Inc.), Goondiwindi 26-27 August 1999.

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## Trap cropping attractiveness and performance.

Martin Dillon

CSIRO Entomology, Australian Cotton Research Institute, Narrabri.

### Introduction

Trap cropping for *Helicoverpa* is becoming increasingly prevalent within the Australian cotton industry. Trap cropping is but one option amongst a broad range of IPM practices that are being used within areawide management strategies. At present trap crops are an unproven technology, and many questions regarding their deployment and subsequent effectiveness remain to be answered.

The principal function of a trap crop is to act as a diversionary host for key pests, attracting and concentrating the pests and their eggs into a relatively small area where the population can be controlled (Hokkanen 1991). If this is successful, pest densities on the primary crops are reduced, resulting in an overall reduction in pest management costs in the surrounding fields. Because trap crops are not necessarily destined for harvest, growers may choose to use slower acting but less disruptive biological pesticides like NPV virus, thereby reducing selection pressure on conventional chemistry, and potentially conserving beneficial insects. Trap crops may also be slashed and cultivated to prevent *Helicoverpa* pupae from surviving. For highly mobile pests such as *Helicoverpa* which readily fly over multiple farms, and that actively choose where they lay each of their many eggs, trap crops would appear to be a logical strategy. This paper reports on some preliminary trials in which we evaluated the impact of trap crops on *Helicoverpa* egg densities in the surrounding cotton in Northern NSW.

### Background

Trap cropping strategies have been successfully employed in cotton production systems against Lygus bugs (*Lygus hesperus*, *L. elisus*) using Lucerne (=Alfalfa) in North America (Stern *et al.* 1969); against Mirid bugs (*Creontiades dilutus*) using within field Lucerne strips in Australia (Mensah and Khan 1997); and against Cotton boll weevil (*Anthonomus grandis*) using early fruiting cotton as a trap crop comprising 5% of the total cotton area (Hardee, 1982). Heliothis have been targeted with experimental trap crops of sesame in Mississippi (Laster and Furr 1972, Pair *et al.* 1982), and with grain sorghum in Oklahoma (Robinson *et al.* 1972). Trap crops of Maize have also been suggested as a potential means of controlling Corn earworm (*Heliothis zea*) in cotton (Lincoln and Isely 1947, Salazar and Martinez 1982)

Trap cropping for *Helicoverpa* in Australian cotton systems was first introduced on a large scale basis in Central Queensland during spring and summer 1997/98 using chickpea and pigeonpea. Dr. Richard Sequeira (QDPI) conceptualised the strategy after demonstrating that the *Helicoverpa* problem in CQ was largely cyclical in nature, being perpetuated by commercial chickpea crops in spring and cotton in summer. The majority of irrigated cotton growers in the region adopted the trap cropping strategy for two main reasons. Firstly, *Helicoverpa* were becoming increasingly difficult and expensive to control due to rising insecticide resistance levels. Secondly, the National Registration Authority viewed trap crops as a necessary component of the INGARD management strategy for CQ.

The Central Queensland trap cropping strategy involves a spring flowering chickpea crop, followed by summer flowering pigeon pea crops. Host plants are generally much more attractive to *Helicoverpa* when they are flowering, and ideally the trap crops remain more attractive than nearby cotton at critical times. The total area planted to trap crops was approximately 1% of the cotton area. This is an arbitrary figure, and there is no evidence to show whether it is sufficient. However one advantage of a small area of trap crops while the technology is being trialed is that it minimizes the risk of excess moths being generated should the trap crops become nurseries. In the 1998/99 cotton season, trap crops were also trialed on the Darling Downs (Chickpea only) and in Northern NSW (Chickpea, pigeonpea

At this early stage the effectiveness of trap cropping for *Helicoverpa* in Australian cotton regions is uncertain. Although high densities of eggs have been recorded in chickpea and pigeon pea trap crops, there has been little or no evaluation of corresponding egg densities in surrounding cotton. The question of whether or not trap crops are economical remains to be answered. Factors that need to be considered include the proportion of eggs diverted away from cotton; the potential lost opportunity cost of the reduced cotton production area; the extra management costs associated with sowing, monitoring, and managing the trap crops; and the costs and effectiveness of controlling pests within the trap crops.

There are also some potential risks with trap cropping strategies that perhaps require consideration. For example trap crops may have the potential act as nurseries that generate unacceptable numbers of pupae and moths. It is also conceivable that some trap crops could act to attract and shelter adult moths that may otherwise have left an area, and which may subsequently lay eggs on the surrounding cotton. Adult moths of both sexes need places in which to feed on nectar and in which to shelter during the day. Finally, there is some experimental evidence that female moths may tend to lay eggs on the types of host plants that they encounter most often (Cunningham *et al.*, 1998). If this 'learning' behaviour is expressed in real field conditions, it may have implications for the effectiveness of small areas of trap crops in diverting eggs away from cotton, the predominant host crop.

### Theoretical considerations

For trap crops to work, they must be more attractive as oviposition sites than the surrounding cotton, so that they attract substantially more eggs per unit area. The trap crops must also occupy a sufficiently large proportion of the cropping area to ensure that a substantial proportion of female *Helicoverpa* moths encounter them during their search for oviposition sites. The minimum effective area for trap crops is unknown. The drawing power of individual trap crops is unknown. The optimum layout of trap crops is also unknown, and our understanding of moth flight behaviour is limited. Strips of trap crop may have the advantage of maximising their perimeter relative to their total area, hence increasing the chances that a randomly moving moth will encounter them. Blocks have less perimeter per unit area, but may have the advantage of allowing moths that encounter them an extended opportunity to lay eggs before they happen to fly outside of the block.

An insight into the potential interaction between trap crop attractiveness and the proportional area sown to trap crops within a region is given in Figure 1. The predicted percentage of total eggs that will be laid on trap crops are plotted for five different amounts of trap crop area (1%, 2%, 5%, 10%, 20%) over a broad range of attractiveness levels. This figure presents theoretical data that assumes that the trap crops are distributed evenly throughout a region, and that moths fly randomly throughout the region. The attractiveness values on the horizontal X axis should be interpreted as follows: Given a free choice between trap crop and cotton, for every egg a female moth lays on cotton, she will lay X eggs on the trap crop. For example, in order to divert 10% of eggs away from cotton and onto an area of 1% trap crops, the trap crop would need a relative attractiveness of 12 eggs per unit area for every 1 egg laid on cotton.

At present there are no quantitative measures of the relative attractiveness of various crops to female *Helicoverpa* moths under field conditions. Qualitative evidence suggests that some crops can be highly attractive to ovipositing females compared with cotton. These include flowering sorghum and silking maize (both for *H. armigera* only), and pigeonpea and chickpea. It is also possible that trap crops may become substantially more attractive in a relative sense, following sprays on surrounding cotton using insecticides that have a strong oviposition deterrence effect. For example pyrethroids are known to deter egg laying for a few days after application. Some food sprays also appear to act as *Helicoverpa* oviposition deterrents.

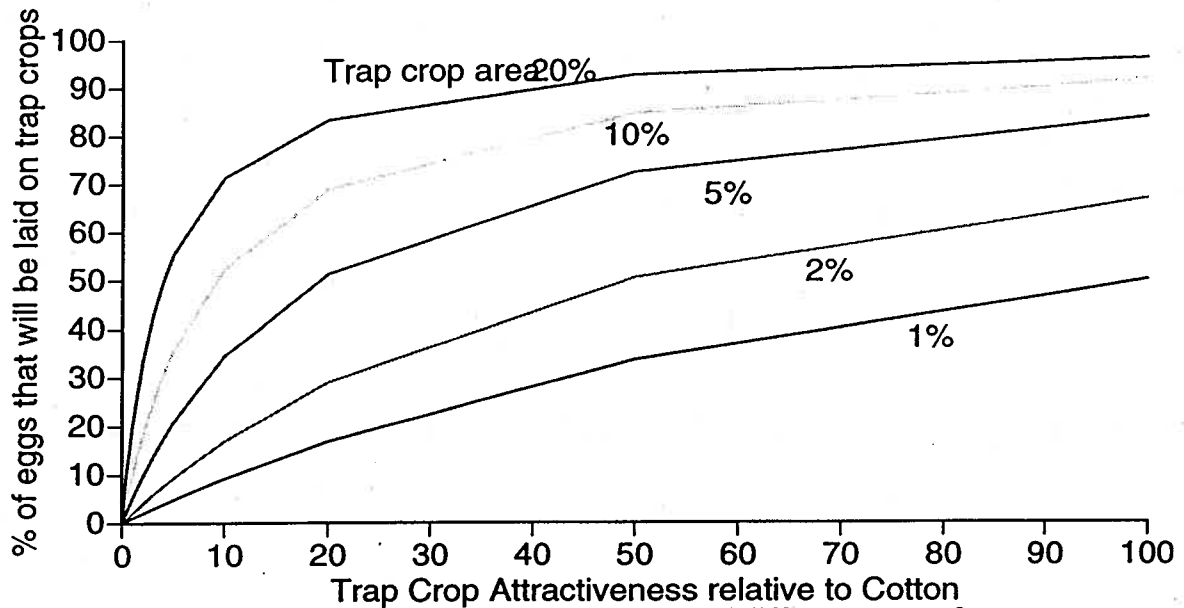


Figure 1. The percentage of eggs laid on trap crops for 5 different areas of trap crop over a range of trap crop attractiveness levels.

Another consideration is the question: What proportion of total eggs need to be diverted onto trap crops in order to begin saving control applications on nearby cotton? This is difficult to quantify with available data. However a preliminary estimate has been made following an analysis of scouting data from 1982-1993 for Auscott Narrabri from the SIRATAC database. Because white eggs do not have thresholds in the Siratac sampling scheme, the analysis centres on VS+S larvae instead. Only checks that exceeded the threshold of 2 VS+S larvae per metre were included in the analysis. The percent reduction in pest density required to bring each count below the threshold was calculated. The percent reductions were then plotted in a frequency distribution, illustrated in Figure 2.

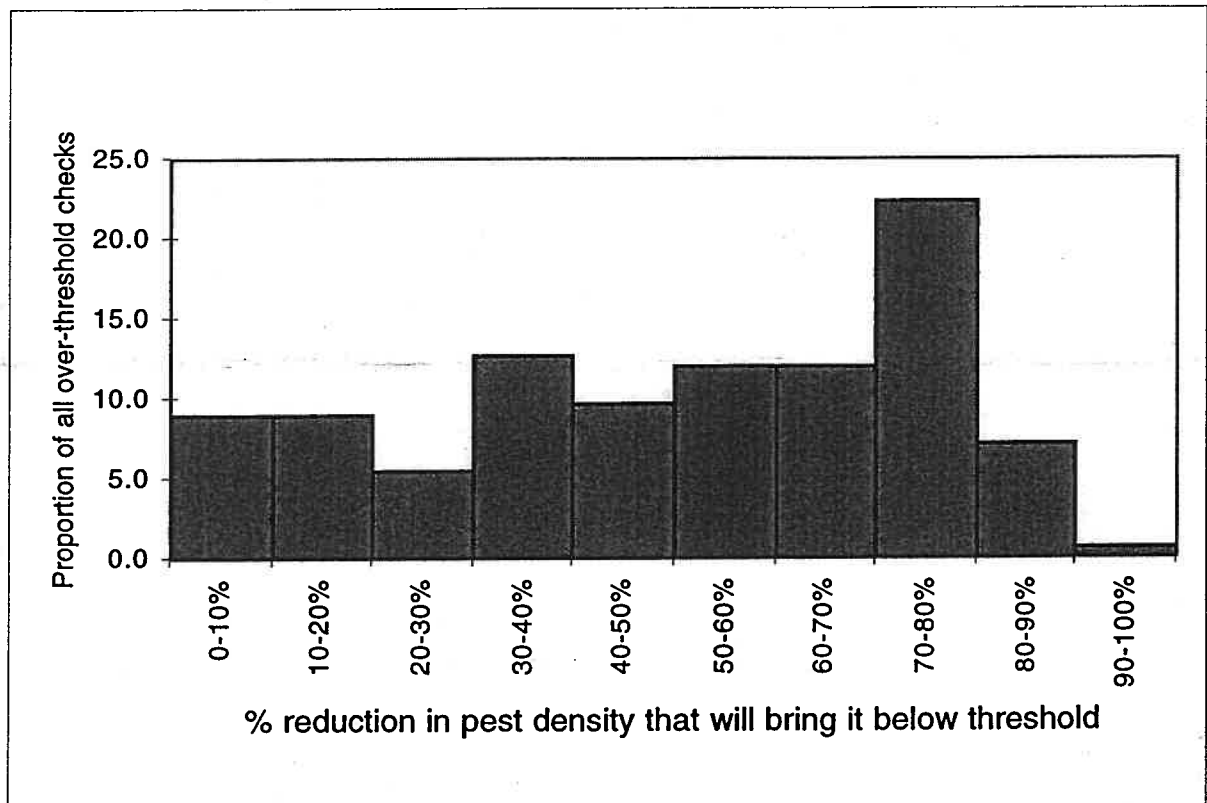


Figure 2. Frequency distribution of the percentage reduction required to bring over-threshold checks in the VS+S larvae counts to below threshold levels.

In the 11 year data set analysed for Figure 2, the proportion of scouting checks over threshold in the VS+S category was 24.8%. The over threshold counts ranged from 2 to 20 VS+S larvae per metre. The distribution of counts between these values was relatively even. Likewise the distribution of percentage reductions that would be required to get the counts below threshold was also even. This would suggest that trap crops are not going to be a "silver bullet". A large proportion of scouting checks that exceed the threshold do so by such a large amount that they would require very substantial reductions in pest density to bring them below threshold. However a reasonable proportion of scouting checks exceed the threshold by only a small amount. For example 9% of the over-threshold checks had counts that could have been brought below threshold if pest densities had been reduced by 10%. The implications of this are that if trap crops could reduce overall egg densities on cotton by for example 10%, then up to 9% of the counts that previously may have been over threshold in the VS+S category would be reduced to below threshold levels. Such a reduction in over-threshold checks is not fantastic, but it may still represent a welcome saving in control costs.

### Pilot study observations on northern NSW trap crops 1998/99

Three areas in northern NSW trialed trap crops for *Helicoverpa* in 1998/99.

1. The Boggabilla Landcare Group planted spring flowering chickpea, followed by pigeon pea or lab lab in the same locations later in summer. They had a total of 43 Ha of trap crop within 4815 Ha cotton (approx. 1%).
2. 'Myambla' at Colly Farms planted summer flowering pigeonpea in 4 x 3 Ha blocks amongst 1200 Ha Cotton (= 1% trap crop).
3. 'Gidgee' at Bourke planted 9.6 Ha of Maize using 3 varieties with 10 days difference in flowering between each amongst 955 Ha Cotton.

We undertook a pilot study on the effectiveness of these trap crops. We monitored egg and larval densities, and observed moth activity over the trap crops, and over adjacent cotton at different distances from the trap crops. The scouting data for these areas is also available, but has not been included in this analysis yet.

Our field studies did not commence in time to monitor egg densities on the chickpea trap crops or on the cotton around them. However pupal counts indicated that a reasonable population of *H. armigera* were successfully trapped. This was significant because of the unseasonal early peak in *H. armigera* numbers. There is circumstantial evidence from spring chickpeas on the Darling Downs that they were successfully diverting *Helicoverpa* eggs away from cotton. When the trap crops were destroyed and cultivated, egg pressure on surrounding cotton crops suddenly increased. This may also be the case at Boggabilla, but the cotton scouting data has not been analysed yet. The average date of the first *Heliothis* spray on conventional cotton amongst the nine Boggabilla landcare group farms was 3 December (earliest first spray: 10 November, latest first spray 17 December).

We monitored moth activity levels and egg densities on the pigeon pea and lab lab, and in the neighbouring cotton at Boggabilla and Collarenebri. In general there were no massive egg lays on these types of trap crops in these areas this season. Egg densities tended to be similar or less than the egg densities on the surrounding cotton. Figure 3 illustrates the density of eggs on a field of pigeonpea at Myambla over a two month period from mid-January 1999 to mid-March 1999.

We did not detect any trends of increasing egg density in cotton with distance away from the trap crops. The hypothesis was that trap crops may attract moths that venture close to them, and therefore adjacent cotton may have a gradient of egg densities that is low close to a trap crop, but that increases with distance away from the trap crop as the influence of the trap crop on moth behaviour decreases. Figure 4 presents results from a sequence of 5 separate counts of eggs within a pigeonpea trap crop, and simultaneous egg counts in adjacent cotton at 5m, 50m, 100m and 200m away from the trap crop. No consistent trend in egg densities is evident.

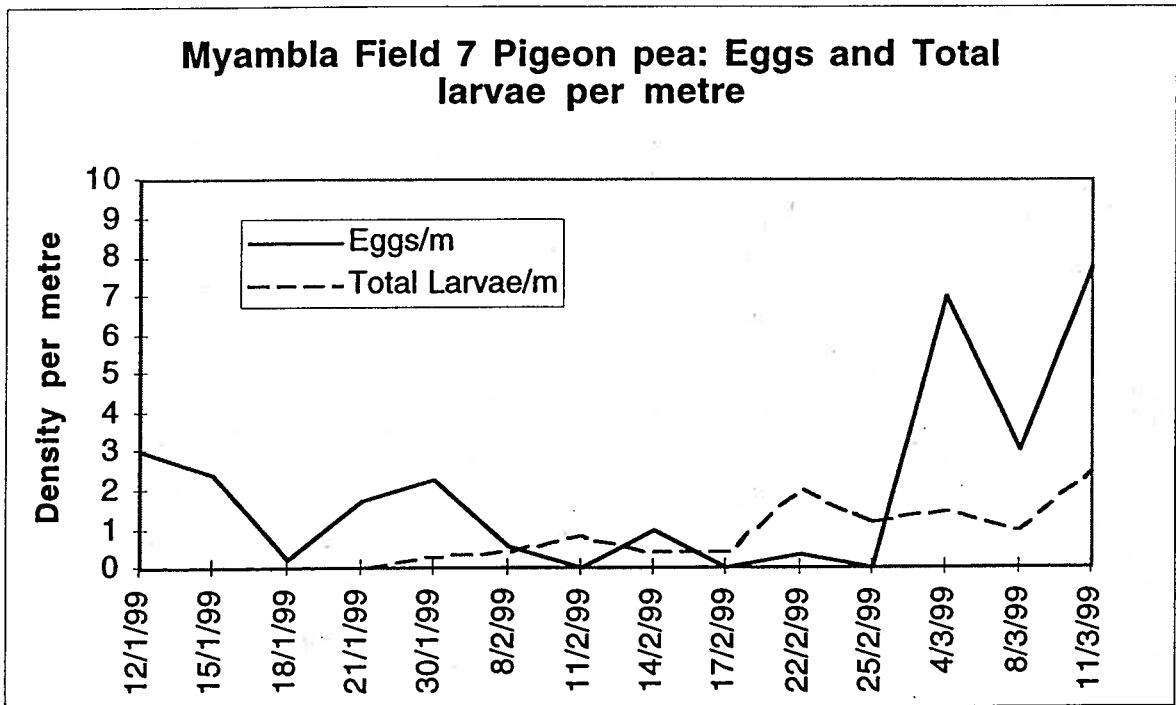


Figure 3. The density of eggs on a trap crop of pigeonpea at Myambla, Colly Farms over a two month period from mid-January 1999 to mid-March 1999

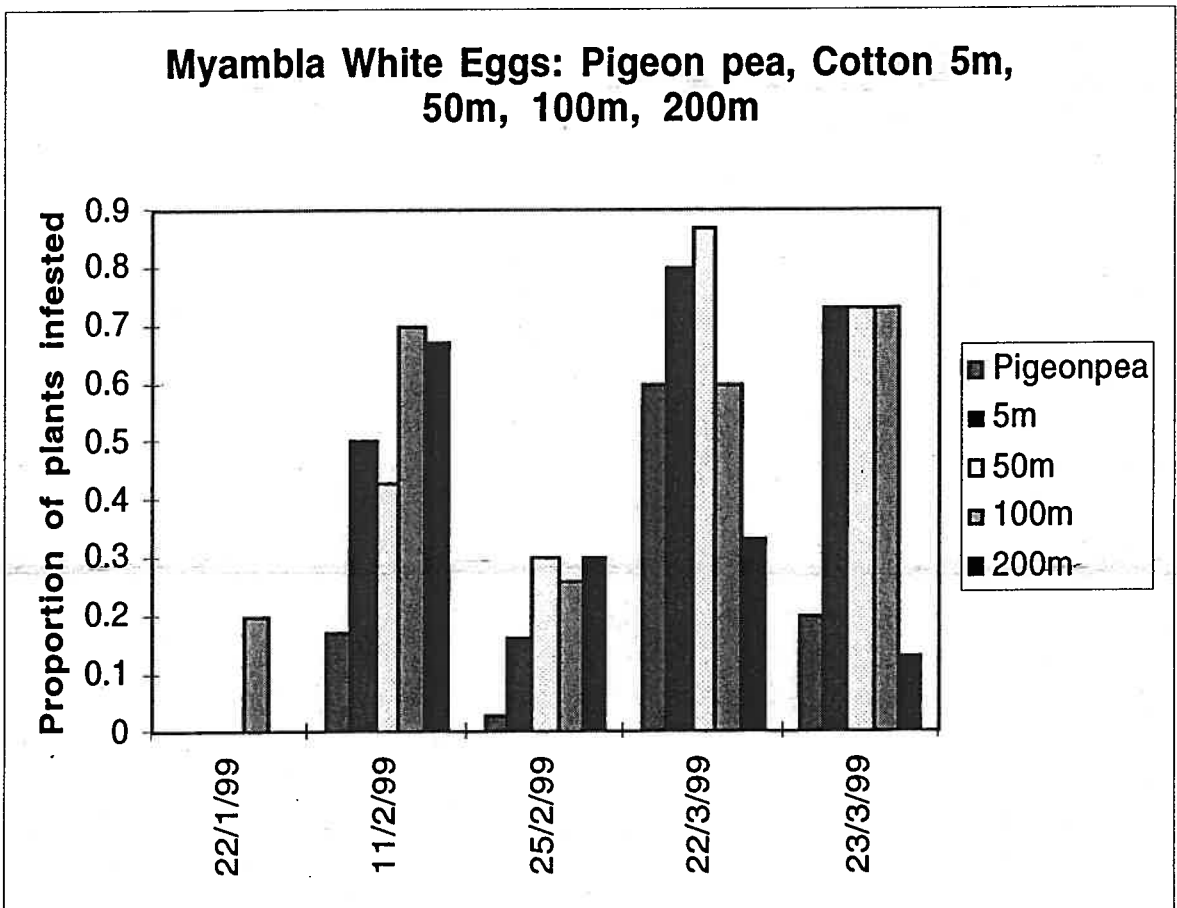


Figure 4. The density of eggs on a pigeonpea trap crop, and on adjacent cotton at 5m, 50m, 100m and 200m distance from the trap crop at Myambla, Colly Farms.

Simon Whyte (NSW Agriculture) monitored egg densities in the cotton around the maize trap crops at Bourke (Figure 5). Egg counts on the maize silks were not recorded. However past experience suggests that egg numbers on the silks could have been substantial, because counts exceeding 200 *H. armigera* eggs per silking cob have been recorded at Narrabri in previous seasons. Egg counts on the surrounding cotton were taken at a number of sites at the following distances from the maize trap crop: 50m, 200m and 400m. Once again, no gradients in egg density were evident with increasing distance from the trap crop.

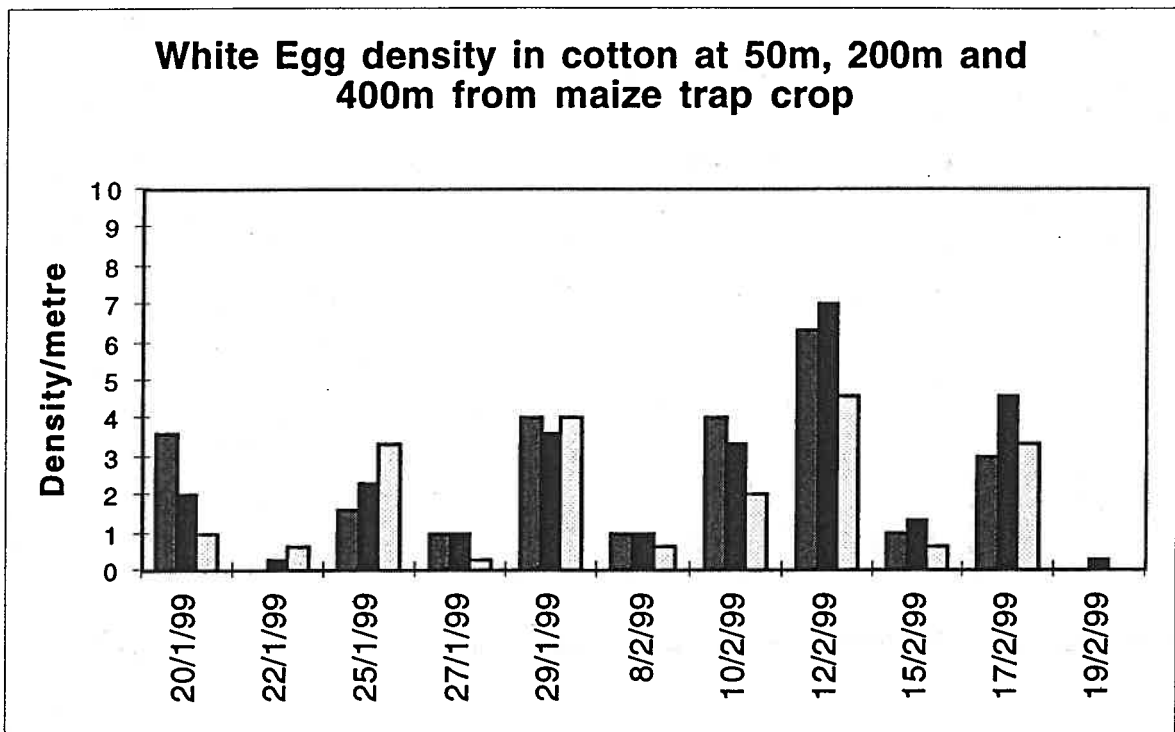


Figure 5. The density of eggs on cotton at 50m, 200m and 400m distance from a trap crop of maize at Gidgee, Bourke.

We also undertook a number of night time observations of moth activity using night vision devices and infra red spotlights (Figure 6). Goal posts were placed within the trap crops, and within the cotton at 5m, 50m, 100m and 200m away. *Helicoverpa* moths passing through the goal posts were counted over 5 minute intervals regularly throughout the first half of the night. These counts provide a relative measure of moth activity, but cannot indicate density. These counts cannot differentiate between *H. armigera* and *H. punctigera*, nor between male and female moths. As for the egg counts, there were no obvious trends in average moth activity levels. We did not observe any difference in the levels of moth activity over trap crops or the surrounding cotton.

Day time flush counts of *Helicoverpa* moths sheltering in the crop canopy were taken by walking along crop rows and counting the number of moths 'flushed' into the open (Figure 7). In general sheltering moths were more abundant in the cotton, perhaps because of the denser canopy. We did not detect any trends in daytime moth numbers with distance away from trap crops.

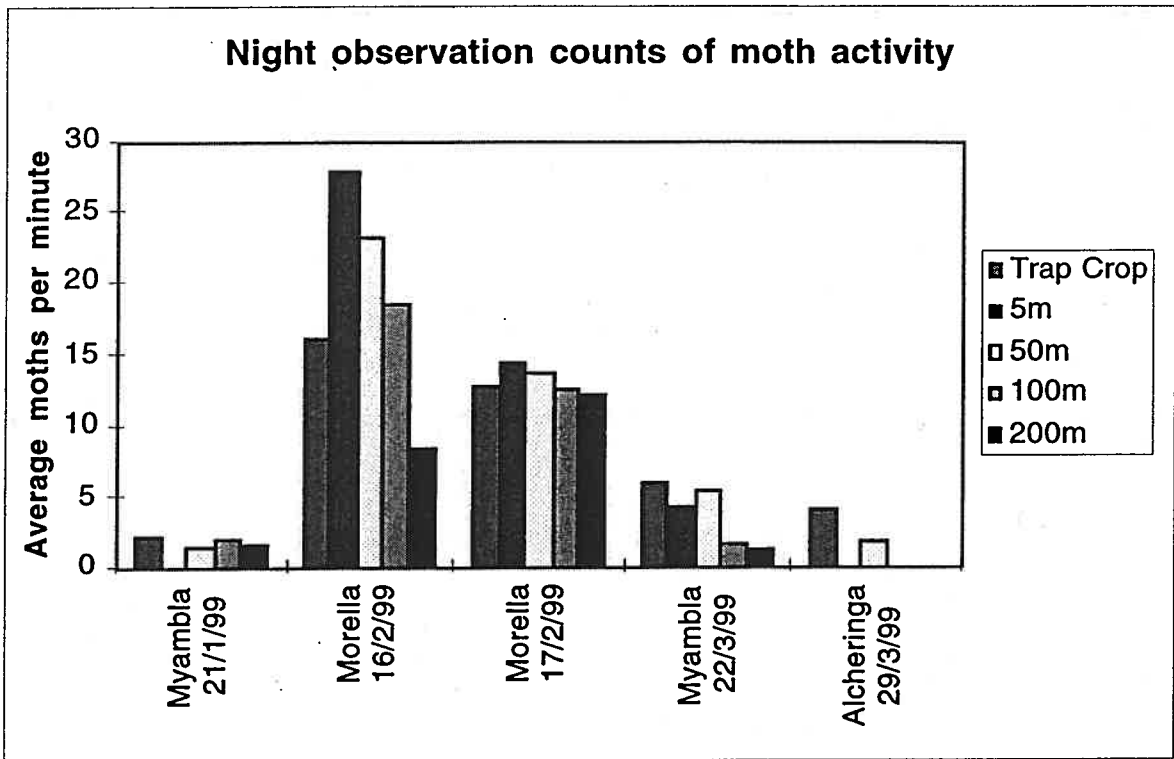


Figure 6. *Helicoverpa* moth activity levels within trap crops, and in adjacent cotton at 5m, 50m, 100m and 200m away from the trap crop.

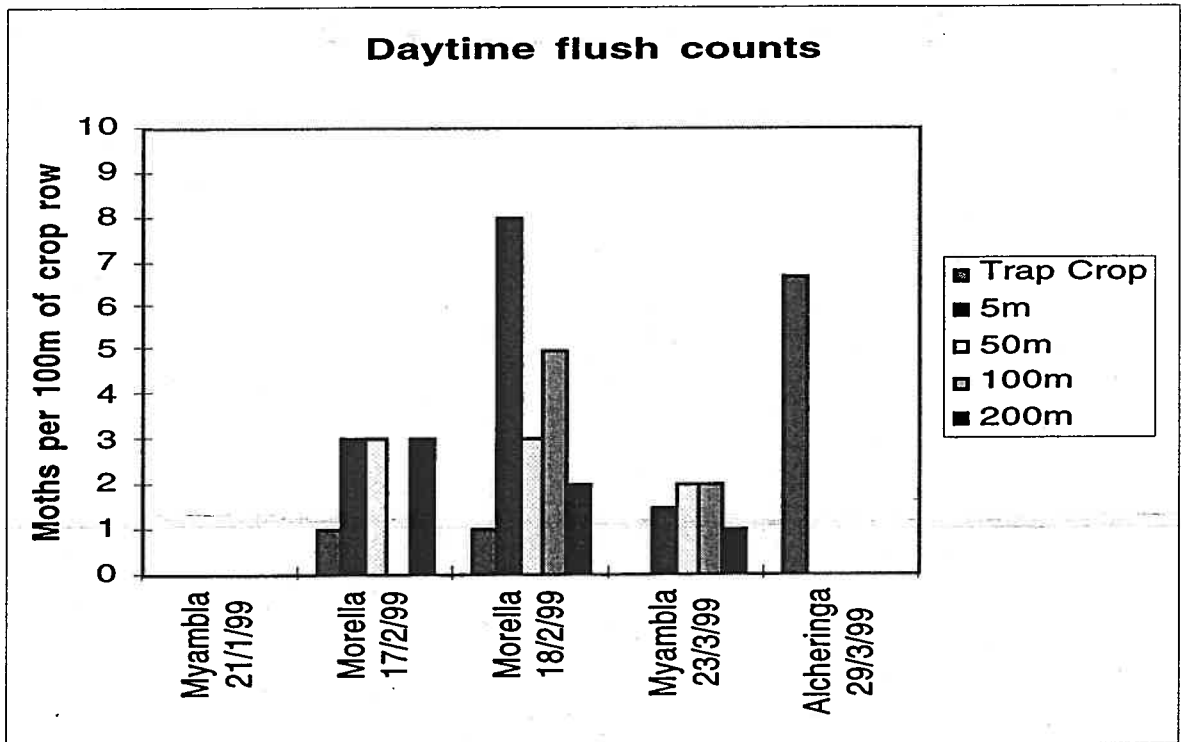


Figure 7. Daytime flush counts of moths per 100m of row in trap crops and in cotton at 5m, 50m, 100m, and 200m distance from the trap crop.

Counts of eggs laid during the night time observations were made the morning after each nights work (Figure 8). As before, there were no trends in egg density related to distance from the trap crop. Furthermore, there were no large peaks of egg densities recorded within the trap crops with one exception. The lab lab field at Alcheringa on 30 March received substantially more eggs than the neighbouring mature cotton. Interestingly, the morning-after

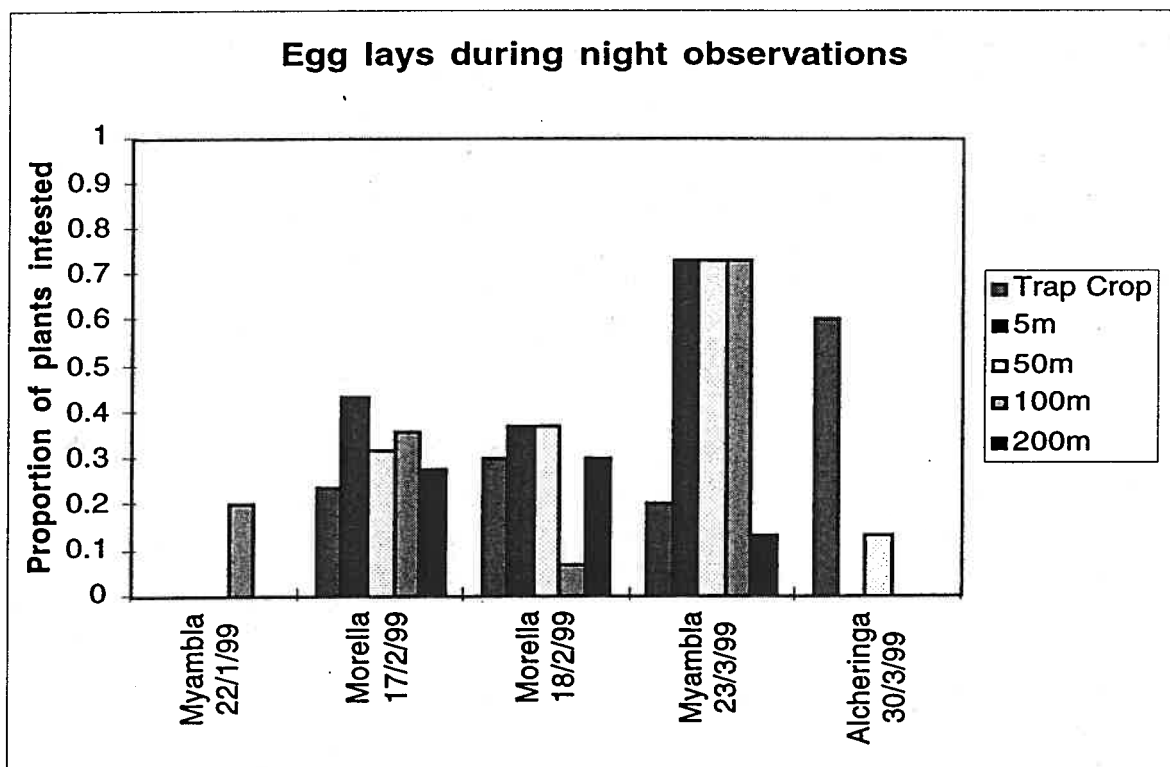


Figure 8. Egg densities following each night time observation in trap crop and cotton at 5m, 50m, 100m, and 200m distance from the trap crop.

## Conclusions

It appears that flowering cotton is a very attractive host for *Helicoverpa*. Mid season trap crops competing with flowering cotton did not perform very well during our pilot study. However when there is a strong contrast in attractiveness between flowering trap crops and non-flowering cotton, as occurs early in the season with spring chickpea or at the end of the season with late lab lab or pigeonpea, the trap crops did perform well. Therefore it may be advantageous to specifically target the early and late season as the best time to have trap crops working in the field. These times have other advantages. Spring trap crops will target the emergence of *H. armigera* from over-wintering pupae. These are the survivors of the previous seasons controls, and tend to be highly resistant to many conventional pesticides. Likewise late season trap crops may act to concentrate resistant *H. armigera* at the end of the season into an area where they can be controlled by cultivation. So as well as the benefit of reducing *Helicoverpa* populations, trap crops also help to combat the *H. armigera* insecticide resistance problem.

For these reasons trap crops for *Helicoverpa* have an important role to play in the Australian cotton industry. There is a very good prospect that if trap crops can reduce egg densities on cotton even by only 5 or 10%, they will save some control costs on the cotton, and help to manage insecticide resistance. However trap crops for *Helicoverpa* are not likely to provide a silver bullet. This is because when *Helicoverpa* exceed thresholds for very small and small larvae, one data set shows that they often exceed thresholds by 20% or more (see Figure 2). The consequences of this are that trap crops would generally need to cause a substantial percentage reduction in pest pressure to bring densities on surrounding cotton below threshold levels.

It is evident that our pilot study was limited in many ways. More data is required that covers a range of seasons and conditions. More data is particularly required on different potential types of trap crops. Research on moth flight and egg laying behaviour under field conditions needs to be undertaken. Many gaps remain regarding the optimum layout and deployment of trap crops. Further analysis of pest pressure and thresholds also needs to be undertaken. Until

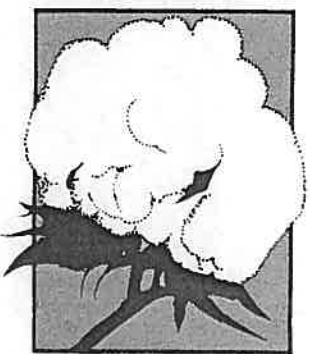
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## Appendix 9

Dillon ML. 2001. INGARD® Refuges v Trap Crops: Are they compatible?  
*The Australian Cottongrower* 22(6): 10-12.

# The Australian cottongrower

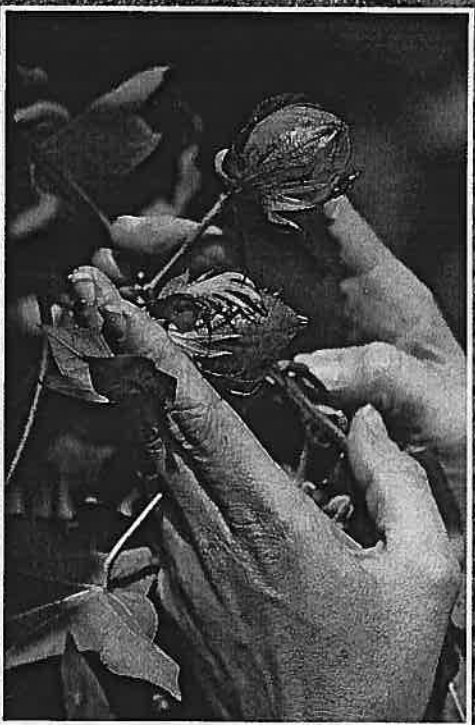


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- **Ingard refuges vs trap crops**
- **Benefits of vetch crops**
- **Cotton on trial at Broome**

# Ingard refuges vs trap crops: Are they compatible?

By Martin Dillon, CSIRO Entomology and Australian Cotton CRC

Whether Ingard refuges and trap crops are compatible is a common question in the Australian cotton industry. This is especially the case with the increased sowing of pigeonpea crops in many valleys — sometimes as an Ingard refuge, sometimes as a *Helicoverpa* trap crop.

The aims behind Ingard refuges and trap crops are very different. A single crop can't perform both functions simultaneously. But separate plantings of pigeonpea refuges and pigeonpea trap crops within a locality and even on the same farm are compatible and can work very well together. The purpose of this article is not necessarily to recommend the combined use of trap crops



with Ingard refuges, but to simply explain the differences between these types of crops and how they can work together.

An Ingard refuge is a crop grown with

the specific aim of generating *Helicoverpa* moths as part of the transgenic Bt cotton resistance management strategy. Ingard refuges produce *Helicoverpa* moths that have not been exposed to Bt toxins at any stage of their life cycle. These moths subsequently emigrate from the refuge and find mates.

The aim is to generate sufficient Bt-susceptible individuals to significantly dilute the chances of two Bt-resistant moths finding and mating with each other. The more moths that an Ingard refuge can produce, the better the dilution will be. So Ingard refuges act as a population source for *Helicoverpa*.

A *Helicoverpa* trap crop is a crop grown with the specific aim of attracting *Helicoverpa* female moths to lay their eggs within it. It concentrates *Helicoverpa* eggs and larvae into a relatively small area where they will eventually be killed. In a well-managed trap crop few, if any, *Helicoverpa* survive to emerge as moths. So trap crops act as a population sink for *Helicoverpa*.

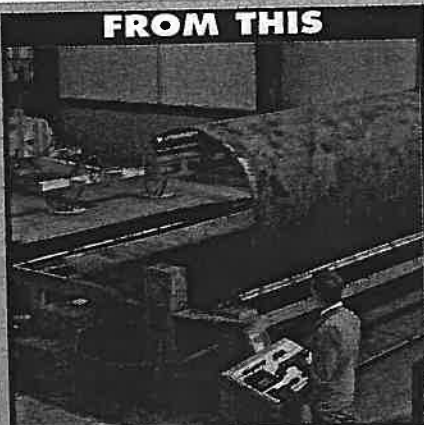
It is crucial that trap crops are closely monitored and destroyed when necessary to prevent them becoming a net source of *Helicoverpa*. Well managed trap crops that act as effective sinks are proving to be very useful tools within IPM and area wide management programs.



A pigeonpea trap crop alongside a cotton crop at ACRI, Narrabri.

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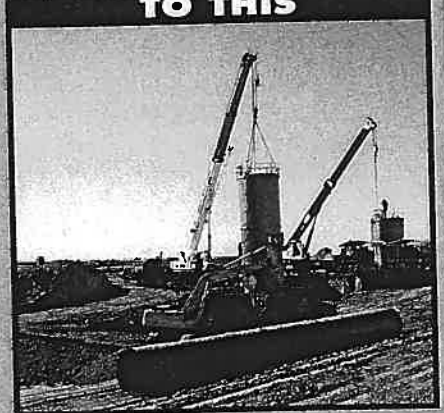
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◀ 10... REFUGES VS TRAP CROPS

Although it may seem strange to be planting Ingard refuges to act as a source of moths on the one hand, and *Helicoverpa* trap crops to act as a population sink on the other (often on the same farm), in fact the technologies can work well together.

Once moths produced by an Ingard refuge have mated with other moths, the refuge's job has been done. Rare resistant genotypes (if they were present) will have been diluted by the proportionally larger number of susceptible genotypes. Those mated moths then have to find somewhere on the landscape to lay their eggs — and what better place to lay them than onto an attractive trap crop?

Some eggs will end up on Ingard cotton — but they will hatch into susceptible larvae that will be killed by the Bt toxin or other control measures applied to these crops. Some eggs will end up on conventional cotton — where they will be subjected to standard management if necessary.

But if an attractive trap crop happens to be present in the vicinity, it will also attract a proportion of the eggs. The larvae that hatch from eggs laid onto trap crops are all doomed to be killed — usually by mechanical means when each trap crop is slashed and cultivated.



**A dead NPV-virus-infected *Helicoverpa* in a pigeonpea trap crop at 'Lowana', Pilliga. *Helicoverpa* in trap crops are killed so these crops act as a sink for the moths.**

In this way trap crops work to help suppress the overall abundance of *Helicoverpa* within a region, and therefore reduce selection pressure for conventional insecticides and for transgenic Bt. Although it is not their primary purpose, trap crops may also act to counteract the effect of production of extra *Helicoverpa* by Ingard refuges.

On a regional scale, Ingard refuges influence the genetic makeup of

*Helicoverpa* populations while trap crops work to influence *Helicoverpa* abundance. While refuges generate moths that do their job by mating with other moths (genetics), trap crops attract eggs and help suppress the overall pest population (abundance).

So the answer to the question. "Are they compatible?" is a definitive yes — Ingard refuges and *Helicoverpa* trap crops work well together. Furthermore, because many refuge and trap crop options use little or no insecticides, they both have the potential to nurture beneficial insects. This increases on-farm biodiversity, and makes the landscape a harsher place for *Helicoverpa* and other pests — an added bonus.

For more information, contact: Martin Dillon, CSIRO Entomology, Narrabri, ph: 02 6799 1518; fax: 02 6793 1186; email: martin.dillon@csiro.au



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## Appendix 10

Heuke L and Dillon ML. (In preparation) Growing pigeon pea as a refuge and trap crop for cotton.

# Growing Pigeon Pea as a refuge and trap crop for Cotton.

Linda Heuke, Research Agronomist (Pulses).  
NSW Agriculture, ACRI, Narrabri.

Martin Dillon, CSIRO Entomology, ACRI,  
Narrabri

## Introduction.

Pigeon pea is an effective trap crop and refuge crop for cotton because it is highly attractive to *Helicoverpa* moths as a host plant for oviposition. During peak flowering, Pigeon pea attracts many more eggs than surrounding cotton crops. Pigeon pea crops are excellent hosts for *Helicoverpa* larvae which feed on the seed pods and flower buds. Unsprayed pigeon pea crops typically produce very high densities of *Helicoverpa* moths.

As part of the Resistance management Plan for INGARD® and Bollgard II® cotton, farmers must plant a refuge crop with the specific aim of generating populations of *Helicoverpa* moths. These moths which have not been exposed to Bt toxins, provide a mating dilution that minimises the possibility of Bt resistant moths mating with each other. Pigeon pea is one of a number of refuge crop options available to cotton growers. It has the advantage that only a small area is required – 5% of the Bt cotton crop area.

Many cotton growers also plant Pigeon pea as a trap crop for *Helicoverpa*. Trap crops have the opposite aim of refuge crops – they aim to kill all eggs and larvae laid within them, and thereby act as a sink that reduces the overall population of moths within the surrounding area.

A single crop of pigeon pea cannot be both a refuge and a trap crop – the goals and management are mutually exclusive. However separate trap and refuge crops of pigeon pea can be grown simultaneously on the same farm, and are compatible with each other.

## The Plant.

Pigeon Pea (*Cajanus cajan* (L.) Millsp.) is a woody, taprooted, weakly perennial, summer growing shrub. Leaves are trifoliolate and leaflets elliptical, 1-2cm wide and 5-8cm long, with pointed ends. Inflorescences are borne at nodes in the upper canopy. Flowers are typical of leguminous plants, about 2.5cm long and consisting of four calyx lobes (the upper two fused) and a large broad standard. Petals are mostly yellow or red, but may also have mottles of purple, cream or other colours. Pods are flat, green to purple, and contain 2-8 seeds

(Troedson 1991). The flowers are attractive to *Helicoverpa* sp. who readily deposit eggs on or near the flowers.

Pigeon Pea is a pulse crop and as such is capable of nitrogen fixation in association with rhizobium bacteria, Group J (CB1024). Levels of nitrogen fixation in the crop are highly variable and the use of N fertilisers may be required for optimal crop growth. Pigeon pea is highly VAM dependent and may require supplementary zinc and phosphorus fertiliser if grown under low VAM conditions.

Australian cultivars (Hunt, Quantum and Quest) tend to be photoperiod insensitive and flower 60-80 days after emergence, with maturity occurring some 60 days after flowering (Troedson 1991). Trials conducted by NSW Agriculture in the Narrabri, Moree and Tamworth districts have resulted in flowers first appearing on cv "Quest" in 70-80 days, plants fully flowering in 75-85 days and plants reaching maturity 170-180 days after sowing. Maturity will be accelerated by cold conditions or water stress.



## Planting.

Pigeon pea should be sown in spring once soil temperatures reach 19°C. Optimal temperatures for plant growth are between 18 and 38°C (van der Maesen, 1989). Pigeon pea refuges must be planted within two weeks of

INGARD® or Bollgard II® cotton crops. Refuge crops. Refuge crops must be planted on the farm unit, within 2 kms of the associated INGARD / Bollgard II fields.

Pigeon pea seeds are a globose to elliptical shape, and cream to brown in colour with some mottling and a seed weight of 0.09-0.14g. They are readily sown using most row crop planting equipment. Aim to establish between 10 to 20 plants m<sup>-2</sup>. A germination test is advisable prior to planting, and seeding rates varied accordingly, as germination can vary widely between seedlots.

#### Herbicide options.

There are several registered chemical options for use pre-plant and post-plant / pre-emergence in pigeon pea. For up to date recommendations please consult the NSW Agriculture publication Weed Control in Summer Crops 2000-2001 (Andrew Storrie, Weeds Agronomist, Tamworth, NSW). "Group A" herbicide options for grassy weed control post emergence are also available. Options for post emergent broadleaf weed control are limited. Pigeon pea grown in an irrigated cotton system gives the option of inter-row cultivation or shielded spraying. The latest information on permits and new registrations is available on the Pulse Australia website ([www.pulseaus.com.au](http://www.pulseaus.com.au)).

#### Soils.

Pigeon pea can be grown on a wide range of soil types varying in pH from strongly acid to alkaline. Zinc deficiency may be a problem on alkaline soils. Pigeon pea is quite drought tolerant, but will not tolerate waterlogging or heavy compaction layers in the soil (Troedson, R.J., 1991). Under irrigated farming systems, the use of raised beds will prevent waterlogging effects. Plants will tolerate saline conditions from 0.6 to 1.2 S/m (Van der Maesen, 1989).

#### Diseases.

No serious diseases of pigeon pea currently exist in Australia. Pigeon pea is susceptible to a form of Fusarium wilt (*Fusarium udum*) however, this is not the same fungus as the cotton infecting form of Fusarium (*Fusarium oxysporum* f.sp. *vasinfectum*). It is therefore highly unlikely that pigeon pea could spread cotton fusarium.

The status of pigeon pea with regard to the hosting and multiplication of the black root rot fungus (*Thielaviopsis-basicola*) is currently unknown. Other pulse crops have been reported to host black root rot (Mondal, A et al, 2000). Until information is available on the

status of pigeon pea, care should be taken if growing pigeon pea in black root rot infected areas.

#### Crop Management.

Pigeon peas planted as a refuge crop must be managed to maintain healthy, attractive plants, and to ensure several cycles of flowering throughout the season.

Pigeon peas are susceptible to frost, and plants will die back naturally as the weather cools. Desiccation may be necessary in warmer areas. Crops used for seed production purposes will need to be protected from insect attack, from budding onwards to ensure quality seed is obtained.

Seed crops are readily harvested using a standard cereal harvester with reduced drum speeds to reduce damage to the grain. Pods are borne on the top of the plant and plants are quite resistant to lodging, so harvest is relatively straightforward.

Refuge crops can be slashed and stalks pulled at the same time as, or after, the surrounding cotton crop is harvested.

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## Appendix 11

Duffield SJ and Dillon, ML. (In preparation). The emergence and control of overwintering *Helicoverpa armigera* in southern NSW.

**The emergence and control of overwintering *Helicoverpa armigera* pupae in southern NSW**

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*Running title: Helicoverpa armigera emergence in southern NSW*

## ABSTRACT

Field trials were undertaken to determine the extent and timing of the spring emergence of *H. armigera* moths from overwintering pupae in southern NSW. A survival rate comparable to that in northern NSW was recorded, suggesting that the colder conditions found further south do not significantly reduce the survival of overwintering pupae. The majority of adults emerged during the last two weeks of October and first two weeks of November, approximately two weeks later than reported in northern NSW. The pattern of emergence is similar to that predicted by the HEAPS model, although the observed emergence tended to be earlier than that predicted by the model. Simulation of the pattern of emergence from 1990 to 2001 predicts a range of approximately four weeks in the date of emergence. This suggests that moths will not start emerging in this part of southern NSW before the beginning of October. The impact of locally used cultivation techniques on the survival of overwintering pupae was also investigated, the results supporting previous findings highlighting the need for either plant line or full cultivation to ensure adequate control of overwintering pupae. Neither direct drilling or lillistons provided consistently reliable control. The results are discussed in terms of the practical management of overwintering pupae in the region.

**Keywords**

cultural control, cultivation, HEAPS

## INTRODUCTION

Significant populations of overwintering *Helicoverpa armigera* (Hübner) (Noctuidae: Lepidoptera) pupae have been found under the crop residues of irrigated summer grain, legume and fibre crops in autumn surveys conducted in southern NSW (Duffield 2004).

The cultivation of crop residues or “pupae busting” is a widely advocated cultural control technique in northern NSW and Queensland (Murray *et al.* 1995, Rourke 2002) and can be undertaken as part of normal farming operations. However before such an approach is adopted in southern NSW the extent of the spring emergence needs to be determined.

Although the majority of fields containing summer crop residues are cultivated during the winter and spring to prepare the ground for the following years summer crop, the first cultivation can be as late as mid-October (Duffield unpublished data). It is therefore important to determine not only if *H. armigera* pupae can survive the winter, but also when the overwintering moths emerge.

The control provided by different cultivation techniques has been extensively evaluated in the cotton industry where the benefits of controlling *H. armigera* pupae by this method have been extensively published (Fitt *et al.* 1993, Wilson 1993, Murray & Titmarsh 1990, Slack-Smith *et al.* 1997). However it is important to evaluate the control gained by cultivation techniques used in the grain industry in the region, to enable relevant advice to be given.

The *Helicoverpa Armigera* and *Punctigera* Simulation model (HEAPS) is a research tool that simulates *Helicoverpa* population dynamics. It contains different modules covering various aspects of *Helicoverpa* population dynamics and their interaction with the cropped environment (Fitt *et al.* 1992, Fitt *et al.* 1995). The diapause emergence module uses the biothermal development model of Cunningham *et al.* (1981) to predict the cumulative proportion of pupae that have emerged. The parameters for the model were derived from field cage data on the Darling Downs in

southern Queensland (Murray 1991). Using local weather data the model can be used to predict the timing and pattern of emergence of *Helicoverpa* in different situations.

This paper describes field experiments conducted to determine the extent and timing of the spring emergence of *H. armigera* in southern NSW and compares the pattern of emergence in the field to that predicted by the HEAPs model. The paper also investigates the impact of locally used cultivation techniques on the overwintering survival of pupae.

## **MATERIAL AND METHODS**

Field experiments were undertaken in 1997 and 1998 in the Riverina region of southern NSW. In each year randomised block trials were undertaken to evaluate the survival of overwintering *H.armigera* pupae under different cultivation techniques and the date of emergence of any surviving pupae.

### **Emergence cages**

2 x 3 m (6 m<sup>2</sup>) tents constructed of white 50% shade-cloth (Sarlon™) were used as emergence cages (after Murray 1995). The tents were 1.0 m in height at one end and 0.75 m at the lower end producing a sloping ridge. A collecting vial was attached at the higher end. Emerging moths move to the highest point of the tent and enter the collecting vial through a mesh cone with a 1.0 cm hole at its apex, the cone then prevents their escape. The trials were monitored on a weekly basis until the first moths were recorded; monitoring then was increased to twice weekly intervals. Monitoring was discontinued when no additional moths were caught during over a two week period. On each sampling date the number, sex and species of moths caught was recorded. The large surface area of the cages enabled them to be used at low pupal densities.

The tents were assembled across the raised beds, and care was taken to exclude areas with wheeling damage. The tent perimeters were dug in to a depth of 10 cm to prevent emerging moths from escaping.

### **Treatments**

In both years a pupal survey of the experimental site was conducted in the autumn to determine pupal densities. Ten 1 m<sup>2</sup> quadrat samples were taken in a W-shaped sampling transect (method described in Duffield 2004)

#### *Experiment 1 - 1997*

A randomised block experiment with four cultivation treatments and three replicates was conducted at Carathool, NSW from May to December 1997. A field of late season maize was selected in April 1997 due to the presence of overwintering pupae. Three soil cultivation treatments were carried out in June 1997.

These were; direct drill (direct drill discs), plant line cultivation (2 ripper tynes and listing rig) and full cultivation (3 ripper tynes and listing rig), a fourth treatment consisted of an uncultivated fallow. The cultivated treatments were subjected to a single pass of the tractor and rig. On all treatments the stubble was mulched and burnt after harvest.

Twelve emergence cages per treatment were erected in the first week of September 1997 and removed in December.

#### *Experiment 2 - 1998*

A randomised block trial with five cultivation treatments was conducted at Whitton in the Riverina, NSW from September to December 1998. The field was selected as it contained a high density of overwintering pupae in April 1998 following an irrigated summer sunflower crop.

Four single-pass cultivation techniques were undertaken in August 1998, these were direct-drill (sod sower), cultivation (lillistons), plant line cultivation (2 ripper tynes + list) and full cultivation (3 ripper tynes + list). Twelve cages per treatment were erected in September 1998 and removed in December.

#### **Data Analysis**

To determine treatment effects an ANOVA was conducted on cage count data. The Bonferroni method for calculating simultaneous confidence limits was then used to distinguish treatment effects (MathSoft 1997).

#### **HEAPS simulations**

The results from both experiments were compared to the predictions of the HEAPS emergence model run using daily maximum and minimum temperature data gathered from the Australian Bureau of Meteorology's climate recording station at Griffith Airport, NSW. Predictions of the spring emergence between 1990 and 2001 were also made using weather data from this site.

## RESULTS

### Experiment 1 - 1997

#### *Pattern of emergence*

The first female moth emerged on the 10<sup>th</sup> October and the first male was recorded two weeks later on the 24<sup>th</sup> October (Figure 1a). The peak rate of emergence of females recorded during the last two weeks of October, whilst peak emergence of males was recorded in the first two weeks of November. Over 50% of the females had emerged by 28<sup>th</sup> October whilst this level was reached by the males on the 7<sup>th</sup> November. The rate of emergence declined from mid October with all moths of both sexes emerging by the fourth week of November.

When compared to the emergence predicted by HEAPS the overall shape of the emergence curve was similar, however the HEAPS prediction was approximately two weeks later than that observed in the experimental cages (Figure 2a). [Consider a "test of parallelism" that compares slopes and a test to compare the intercepts. Perhaps we may need to transform the data (log?) to straighten the lines for direct comparison]

#### *Effect of cultivation of survival*

A mean of 1.6 diapausing pupae were recorded per square metre in the autumn pupal survey, which equates to 9.6 pupae per cage. A mean total of 2.3 moths were recorded per cage in the uncultivated control, representing a survival rate of 24%.

Significant differences ( $F_{3,44}=7.06$ ,  $P<0.001$ ) were detected between the different treatments. The numbers of moths recorded in the uncultivated control differed significantly from the plant line ( $P<0.05$ ) and full cultivation ( $P<0.01$ ) treatments, but did not differ significantly ( $P>0.05$ ) from the direct drill treatment (Figure 3a)

### Experiment 2 - 1998

#### *Pattern of emergence*

In 1998 female moths started to emerge in the second week of October (13<sup>th</sup>) with 50% emergence taking place by 6<sup>th</sup> November (Figure 1b). Male moths were first recorded on 23<sup>rd</sup> October with 50% emergence not occurring until the 17<sup>th</sup> November. No further emergence was recorded after 11<sup>th</sup> December.

The emergence predicted by the HEAPS model was slightly later to that observed in the experimental cages (Figure 2b) although the difference was less than that observed in 1997.

The predicted overall pattern of emergence in 1997 and 1998 were initially similar, but diverged after November 17. This divergence was caused by warmer daily temperatures after this date in 1997 compared to 1998. This resulted in the cumulative emergence of moths reaching 99% in 1997 11 days earlier than in 1998.

#### *Effect of cultivation on survival*

A diapausing pupal density of 18.1m<sup>-2</sup> was recorded in the pupal survey conducted in the experimental site in May 1998, representing 108.6 pupae per cage. The mean number of moths emerging in the uncultivated treatment was 9.8 representing a survival rate of 9.0% (Figure 3b).

Significant differences ( $F_{4,55}=19.64$ ,  $P<0.001$ ) were detected between the treatments. All the cultivation techniques differed significantly ( $P<0.01$ ) from the untreated fallow. The treatment with Lillistons differed significantly ( $P<0.05$ ) from the other cultivation techniques. Less than 2 moths per cage were recorded from the plant line cultivation and full cultivation treatments.

#### **Long term predictions of spring emergence from diapause**

The predicted patterns of emergence over 12 seasons, and pattern predicted from the long term average (1990-2001) are presented in Figure 4. The predicted emergence for 1997 and 1998 are both relatively close to the long term average, in comparison to the range of predicted patterns of emergence for other years. The simulations suggest

a range of approximately four weeks in the timing of 50% emergence depending on the weather conditions. [Consider plotting and describing this as an average bounded by 95% confidence intervals, rather than 12 separate lines ?]

## DISCUSSION

The results of these experiments indicate that *Helicoverpa armigera* is able to successfully overwinter as pupae in southern NSW. The recorded survival in uncultivated situations of between nine and twenty-five percent is comparable to that recorded in northern NSW (Fitt & Daly 1990), suggesting that the colder conditions found further south do not significantly reduce the survival of overwintering pupae.

The finding that females tend to emerge several days before has been reported previously for *H. armigera* (Foley 1981, Murray 1991). This is an evolutionary adaption, ensuring that the sexes are in a reproductive stage of development simultaneously.

The emergence of adults from overwintering pupae is later than reported in northern NSW by approximately two weeks. This would be expected due to the cooler spring temperatures found in this region. The observed difference between the HEAPS prediction and the field results could be due to a number of reasons. It may be caused by differences in the experimental design of these experiments from the calibration trials. For example although the cages in this study were based on the same design as those used to calibrate the HEAPS model, the material used may have differed leading to differences in the temperature within the cages. Conversely the discrepancy may be due to differences in the soil type. The thermal properties of the soils in the Riverina could differ from those where the model was calibrated, for example they may warm faster in spring, or the physical structure could differ leading to the pupae being found at different depths to those in the calibration study. Murray (1991) reports that for every 1mm increase in pupal depth, emergence of *H. armigera* from diapause is delayed by 0.78 days. The causes of the difference observed in this study were not investigated further, however this finding does suggest that field calibration of the HEAPS model would be required before the model was used to predict the emergence of *H. armigera* in regions distant from the area where the model was calibrated.

The HEAPS simulation of the years from 1990-2001 predicts a range of about four weeks in emergence date depending on climatic conditions. Building in the observed discrepancy between the model and the field observations this still means that

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*H.armigera* moths will not start emerging in this part of southern NSW before the first week of October. This pattern of emergence provides a longer window of opportunity for pupae busting compared to the regions to the north.

The effect of cultivation has been evaluated in the cotton industry where the benefits of controlling *H. armigera* pupae in this way have been extensively published (Fitt *et al* 1993, Wilson 1993, Murray and Titmarsh 1990, Slack-Smith *et al.* 1997). The results of this study support previous findings highlighting the need for either plant line or full cultivation to ensure adequate control of overwintering pupae. Neither direct drilling or lillistons provide consistently reliable control.

This study has identified the date in spring by which control needs to be undertaken and the type of locally practised cultivation required to destroy pupae. Previously reported research (Duffield 2004) has highlighted which fields are likely to contain high populations of overwintering *H. armigera* pupae. When these data are taken together they provide accurate guidance on the best way of managing local populations of *H. armigera* through the targeted cultivation of overwintering stubbles.

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Figure 1. Observed emergence of *H. armigera* males and females in a) 1997 and b) 1998.

Figure 2. Observed and HEAPS predicted patterns of *H. armigera* emergence a) 1997 and b) 1998.

Figure 3. The effect of cultivation on the emergence of *H. armigera* in cultivation trials a) 1997 and b) 1998.

Figure 4. HEAPS predicted patterns of *H. armigera* spring emergence from overwinter diapause over the 12 year period 1990 to 2001.

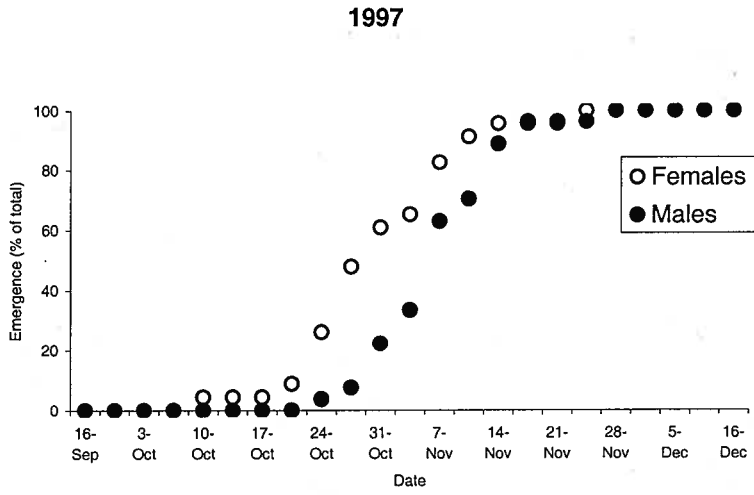


Fig 1 a)

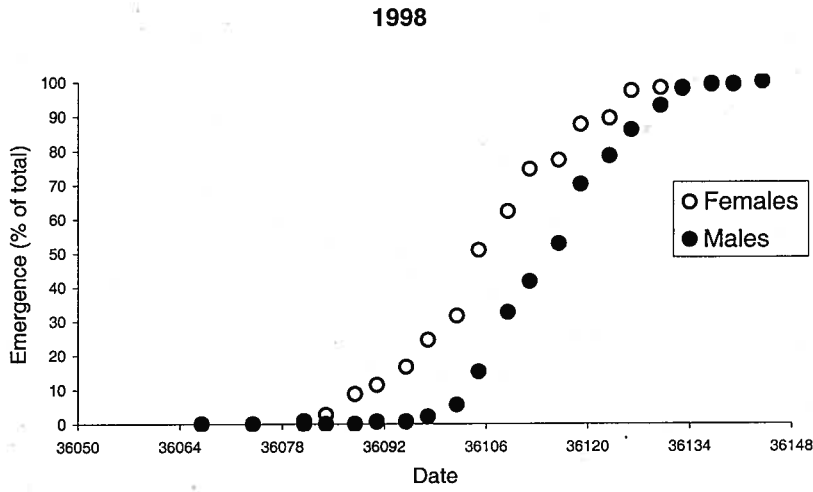


Fig 1 b)

Figure 1. Observed emergence of *H. armigera* males and females in a) 1997 and b) 1998.

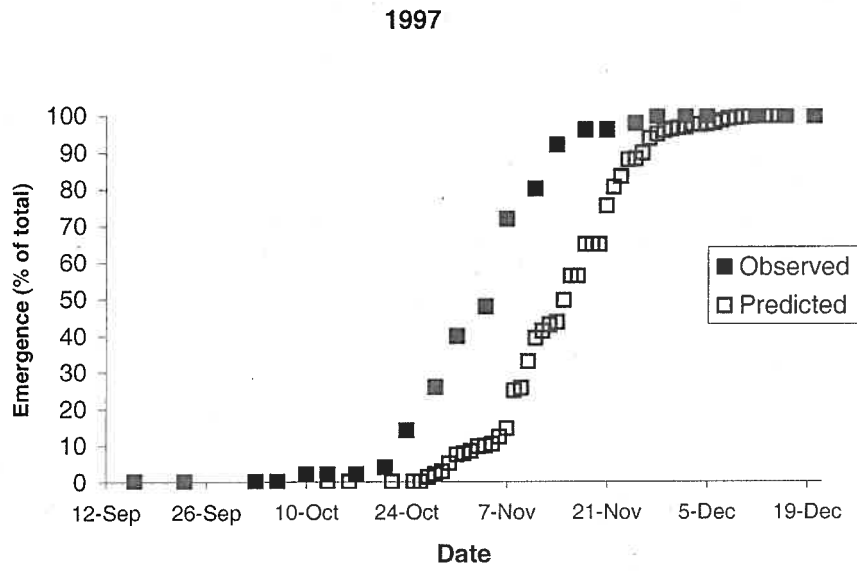


Fig 2 a)

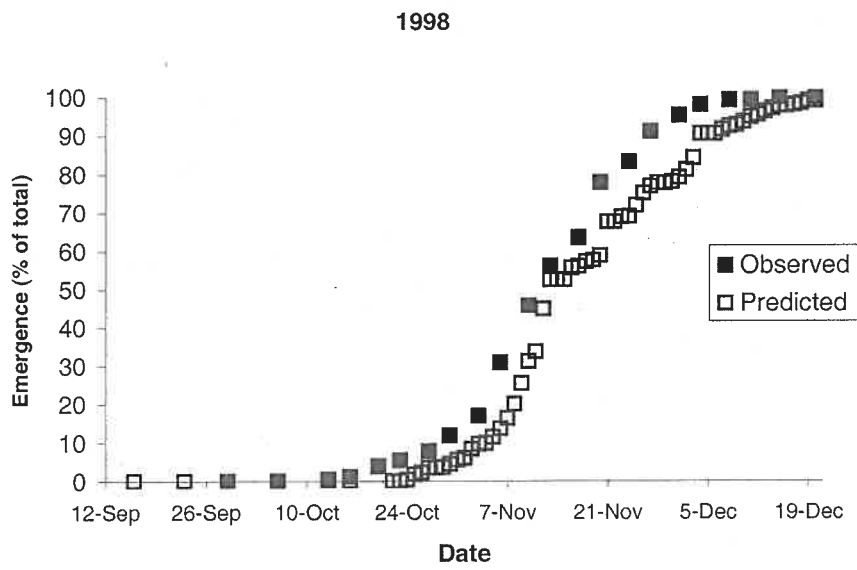


Fig 2 b)

Figure 2. Observed and HEAPS predicted patterns of *H. armigera* emergence a) 1997 and b) 1998

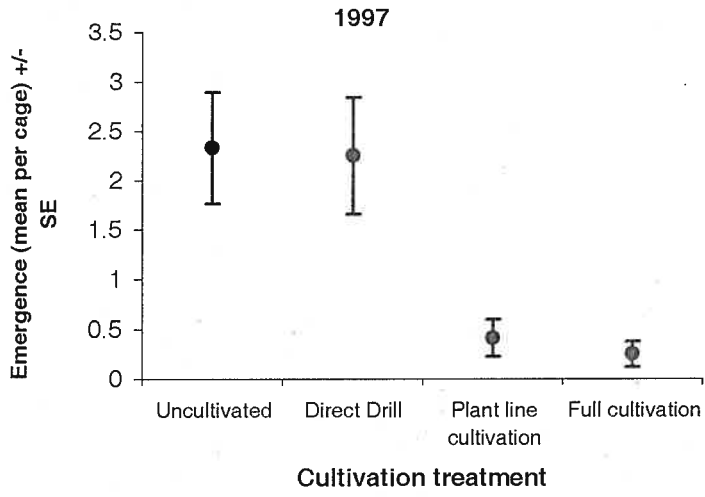


Fig 3 a)

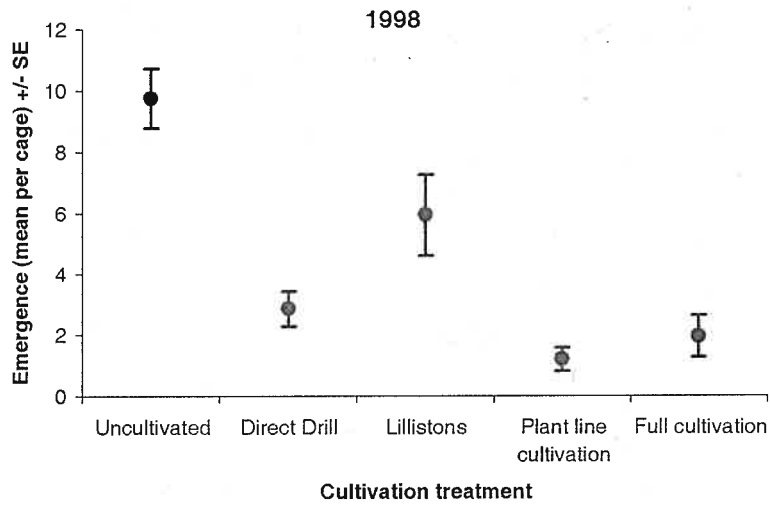


Fig 3 b)

Figure 3. The effect of cultivation on the emergence of *H. armigera* in cultivation trials a) 1997 and b) 1998

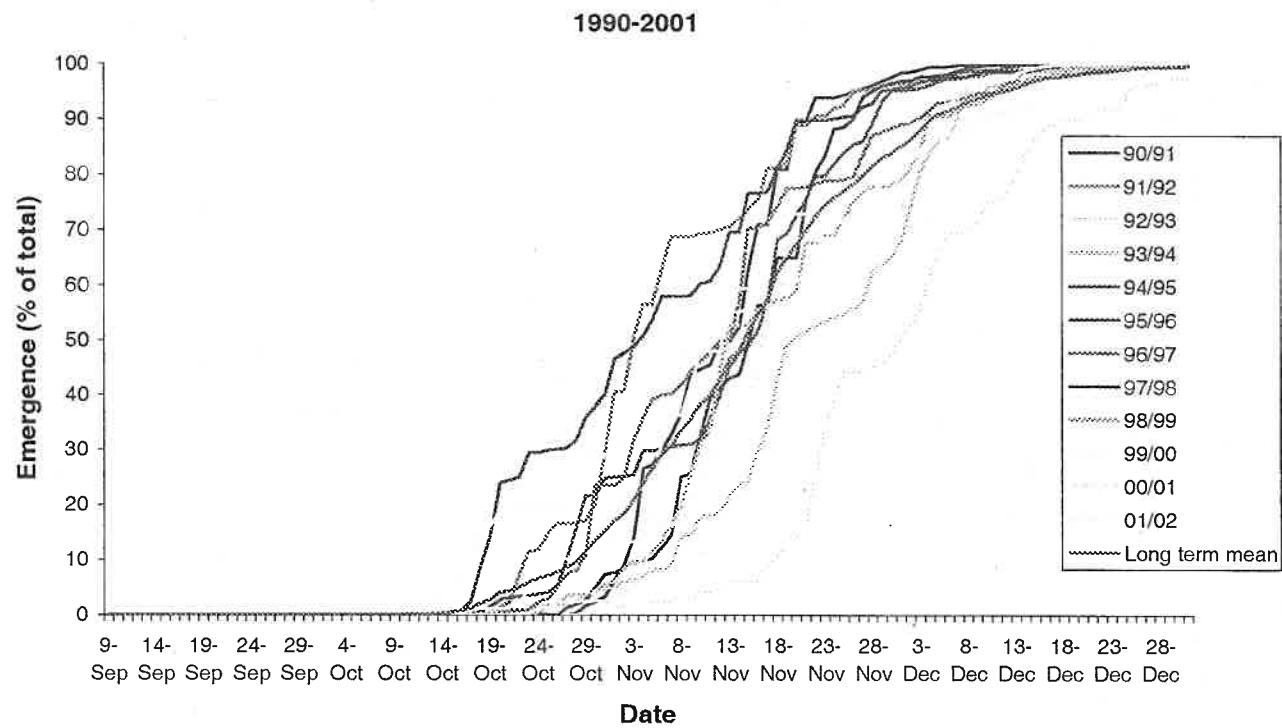


Figure 4. HEAPS predicted patterns of *H. armigera* spring emergence from overwinter diapause over the 12 year period 1990 to 2001.