Table 1. Performance of promising medium duration desi chickpea genotypes against *Helicoverpa armigera* at Varanasi, Uttar Pradesh, India, postrainy season 1992/93–1994/95.

Genotypes	Mean number of pods plant^{-1}	Pod damage (%)			Resistance/ susceptibility ratings			Overall mean of resistance/ susceptibility
		1992/93	1993/94	1994/95	1992/93	1993/94	1994/95	
GL 1014	57.9	20.6 (26.9)[1]	7.8 (16.1)	6.26(14.2)	4	3	3	3.3
PDG 90-2E	66.5	21.4 (27.5)	17.3 (24.5)	13.1 (21.1)	5	6	5	5.3
ICCX730020-11-2-H	63.3	-[2]	15.7 (23.4)	10.9 (19.2)	-	5	4	4.5
AKG 33	62.5	-	28.9 (32.5)	11.1 (19.5)	-	9	5	7.0
BG 373	45.3	27.9 (31.7)	22.3 (28.2)	16.4 (23.8)	6	7	7	6.3
BG 362	39.5	36.6 (37.2)	15.8 (23.4)	13.9 (21.8)	7	5	6	6.0
Pant CE1	51.4	-	14.4 (22.3)	12.7 (20.5)	-	5	5	5.0
Pant CE2	55.0	-	16.7 (24.1)	17.6 (24.6)	-	5	7	6.0
Radhey	57.5	24.2 (29.3)	30.6 (33.6)	22.4 (23.2)	6	9	9	8.0
C 235 (control)	53.0	28.5 (32.2)	19.2 (25.8)	14.7 (22.3)	6	6	6	6.0
H 208	62.0	29.4 (32.8)	21.9 (27.9)	23.3 (28.7)	9	7	9	8.3
BG 256	48.2	-	11.1 (19.4)	12.8 (20.8)	-	4	5	4.5
SE	-	±1.63	±1.20	±1.47	-	-	-	
CD at 5%		(5.0)	(3.4)	(4.2)				

1. Figures in parentheses are arcsine transformed values.
2. Indicates genotypes not tested in that year.

Reed, W., Cardona, C., Sithanantham, S., and **Lateef, S.S.** 1987. Chickpea insect pests and their control. Pages 283–318 *in* The Chickpea (Saxena, M.C., and Singh, K.B., eds.). Wallingford, Oxon, UK: CAB International.

Evaluation of Insecticide Mixtures for Controlling *Helicoverpa armigera* on Chickpea

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Helicoverpa armigera has developed resistance to most of the classes of insecticides recommended for bollworm control in India. Resistance levels are particularly high to the currently available pyrethroids, endosulfan, and at least some of the organophosphates (Armes et al. 1992). As a result of the often poor control of *H. armigera* using single chemicals, Indian farmers frequently tank-mix two or more products in order to contend with high pest pressures on cotton and other crops (Armes et al. 1995). Recently a number of commercial pyrethroid/organophosphate insecticide mixtures have become available. Such mixtures may have the potential to control *H. armigera* populations which are resistant to the individual constituents of the mixture, if a synergistic interaction between the chemicals occurs at the biochemical level. There is also scope to use mixtures of biological insecticides such as *Bacillus thuringiensis* (Bt) with pyrethroid insecticides. Such combinations have proven effective in managing pyrethroid resistant *H. armigera* in cotton in Australia (Shaw 1993).

Poor control with conventional pyrethroid and organophosphate insecticides of heavy infestations of *H. armigera* on chickpea on the IAC farm during Dec 1995, prompted us to set up a simple field experiment to evaluate the performance of two newly introduced commercial insecticide mixture formulations: Spark® (Hoechst Schering AgrEvo Ltd.) and Polytrin-C® (Ciba Geigy Ltd.), and a tank mix of cypermethrin and Delfin WG® (Sandoz Ltd.).

The treatments (unreplicated with the exception of the unsprayed control), were as follows: Spark® (deltamethrin 1% + triazophos 35%), 1 L ha^{-1} Polytrin-C® (cypermethrin 4% + profenofos 40%), 1 L ha^{-1}; Cypermethrin 10% EC, 1 ha^{-1}; Delfin WG® (*Bacillus thuringiensis* s., sp kurstaki), 1 kg ha^{-1}; Cypermethrin 10% EC + Delfin WG®, 1 L + 1 kg ha^{-1}. The plot sizes were 0.5 ha and comprised a range of different chickpea genotypes in each plot. Insecticides were applied once only with an Allman tractor mounted boom sprayer at 325 L ha^{-1}. All applications were made on 28 Dec 1995 when most genotypes where at the peak flowering stage. Counts of *H. armigera* larvae were made on ten randomly selected plants in each plot, at 2–3 hours before spraying and then at 2, 5, 8, and 12 days after application.

The effects of the various insecticide treatments on *H. armigera* larval densities, compared to the untreated control plots are summarized in Figure 1. For purposes of clarity the data are presented on two graphs. On the basis of larval counts, Polytrin-C® and Spark® were highly effective, maintaining larval numbers at 1–2 $plant^{-1}$ from 2–12 days after treatment, compared to the control plots where larval numbers were very high at 8–10 $plant^{-1}$. Bt applied alone, reduced larval numbers to 3–4 $plant^{-1}$ up to 8 days after treatment. No control advantage was observed with the mixture of cypermethrin + Bt; by 12 days after treatment, larval numbers exceeded the numbers recorded in the control plots. Cypermethrin applied alone performed poorly with larval numbers actually increasing from 2–5 days after treatment. Overall there was little or no advantage of applying cypermethrin compared to not using insecticide at all.

The poor performance of cypermethrin for *H. armigera* control on chickpea was not surprising in view of the dynamics of pyrethroid resistance in *H. armigera* in the Hyderabad region in India. Very high cypermethrin resistance frequencies have consistently been reported toward the end of the rainy cropping season, Dec–Mar, over the past 5 years (Armes et al. In press). It is likely that all larval stages, perhaps with the exception of neonates, are immune to recommended field application rates of cypermethrin in the Hyderabad region during the chickpea season. Use of broad-spectrum pyrethroids at this time probably impacts heavily on natural enemy populations, but has little effect against pyrethroid-resistant *H. armigera,* with the result that pest numbers can reasonably be expected to increase. Bt was only marginally effective in controlling *H. armigera* larvae. These data support earlier observations where the performance of Bt products for control of *H. armigera* on cotton were poor, particularly against older larvae (third to sixth instar) (Armes, unpublished data). The levels of control achieved with Spark®, and particularly Polytrin-C®, were encouraging, and clearly show that despite high levels of pyrethroid resistance in *H. armigera* populations, pyrethroid/organophosphate insecticide mixtures can provide an effective means of

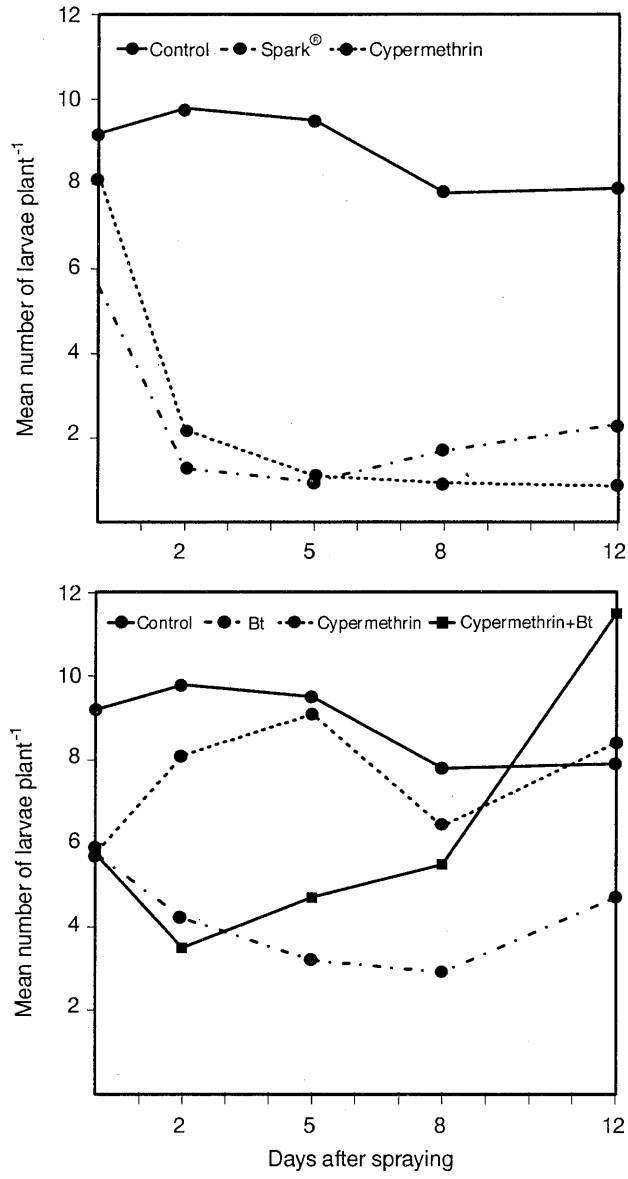

Figure 1. Effects of different insecticides and insecticide combinations on the mean numbers of *Helicoverpa armigera* larvae (all instars), per chickpea plant from 0 to 12 days after application, IAC, Hyderabad, India, Dec 1995–Jan 1996.

controlling this pest. Further trials at IAC are planned for the 1996/97 chickpea season.

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Crop Quality/Utilization

Effect of Germination on Tannin Concentration in Chickpea

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Besides being an important source of protein, chickpea is also reported to be a good source of minerals (Meiners et al. 1976, Jambunathan and Singh 1981). Legumes also contain antinutritional factors which affect digestibility adversely. Tannins, phenolic polymers located in the testa, are produced in many plants as secondary metabolites, and are considered the major antinutritional factors. The antinutritional activity of tannins is caused by complexation with digestible proteins in general, and with digestive enzymes such as trypsin and chymotrypsin which interfere with iron absorption (Reddy et al. 1985). Tannin-free or low-tannin chickpea can easily be obtained by selection. Tannins are higher in chickpea cultivars with a dark seed-coat color (Singh 1987). Chickpea is consumed after processing, including such traditional processes as soaking and sprouting. This study reports the distribution and changes in tannin content during germination of different varieties of chickpea grown in Gulbarga, Karnataka.

Chickpea seeds grown locally were obtained from the Pulse Research Centre, Gulbarga. The chemicals used in this study were purchased from Loba Chemicals, Bombay. Desi and kabuli varieties grown in Gulbarga region were purchased from the local market. Tannin concentration was determined in 1 g of powdered chickpea meal according to the method of Price et al. (1978). Seventeen chickpea genotypes were tested for tannin content. As Table 1 shows, tannin concentration ranged from 0.05% to 0.21%. Genotypes GAS 34 and CGS 11 contained the highest tannin concentration (0.212%) and the variety GAS 38 contained the least (0.054%). The dark-brown seed coat of GAS 34 and CGS 11 indicates their high tannin concentration. This observation agrees with the results of Butler (1982) that high tannin concentration causes the dark pigmentation of testa. Bressani et al. (1984) have also observed lower amounts of tannins in white than in dark, red, and bronze varieties of dry beans. Similar observations have been reported by Singh (1987) for chickpea and pigeonpea. Tannin concentrations were 0–0.2% in pigeonpea (Price et al. 1980) and 0.135–0.68% in winged bean (Kotaru et al. 1987).

The effect of germination of chickpea on the tannin content of five chickpea varieties is given in Table 2. Nearly 50% tannins were lost when chickpea was soaked overnight in distilled water. The decrease in tannin concentration after soaking is because the tannins are leached into the water. Sprouting and germination of chickpea decreased tannin concentration. Similar results have been reported in mung bean, pigeonpea, and chickpea (Reddy et al. 1985). A decrease in tannin concentration was observed in all the five varieties tested, when the germination period was increased. Less than 10% of the original tannin content was observed in three varieties, Kabuli LM, TNG 18, and GAS 10, when germinated for 5 days. The drop in tannin concentration during soaking and germination is because of leaching of the tannins into the water. It may also be because of the activity of polyphenol oxidize and enzymatic hydrolysis.

Our study indicates the presence of tannins in chickpea, and that soaking and germination helps in reducing the levels of tannins.