We are IntechOpen, the world's leading publisher of Open Access books Built by scientists, for scientists

4,800
Open access books available

122,000
International authors and editors

135M
Downloads

Our authors are among the

154
Countries delivered to

TOP 1%
most cited scientists

12.2%
Contributors from top 500 universities

CLARIVATE ANALYTICS BOOK CITATION INDEX INDEXED

WEB OF SCIENCE™

Selection of our books indexed in the Book Citation Index in Web of Science™ Core Collection (BKCI)

Interested in publishing with us?
Contact book.department@intechopen.com

Numbers displayed above are based on latest data collected.
For more information visit www.intechopen.com

Chapter 2

Plutella xylostella (Linnaeus, 1758) (Lepidoptera: Plutellidae): Tactics for Integrated Pest Management in Brassicaceae

S.A. De Bortoli, R.A. Polanczyk, A.M. Vacari,
C.P. De Bortoli and R.T. Duarte

Additional information is available at the end of the chapter

http://dx.doi.org/10.5772/54110

1. Introduction

The diamondback moth, *Plutella xylostella* (L.) (Lepidoptera: Plutellidae), is one of the most serious pests of cultivated Brassicaceae worldwide [1,2]. This crucifer specialist may have its origin in Europe [3], South Africa [4], or East Asia [5], but is now present worldwide wherever its host plants exist [6].

In the first instar, the larvae enter into the leaf parenchyma and feed between the upper and lower surfaces of leaves creating mines. In the second instar, the larvae leave the mines, and from the second to the third instar, they feed on the leaves, destroying the leaf tissue except for the upper epidermis, leaving transparent "windows" in the leaves. Fourth-instar larvae feed on both sides of the leaves [7]. This insect has a short life cycle, around 18 days, and its population may increase up to 60-fold from one generation to the next [8]. Studies indicate that the moths can remain in continuous flight for several days while covering distances up to 1000 km per day, but how the moths survive at such low temperatures and high altitude is not known [1]. In eastern Canada, annual populations of diamondback moths originate from adult migrants from the United States [9].

P. xylostella was the first crop insect reported to be resistant to dichloro-diphenyl-trichloroethane (DDT), only 3 years after the start of its use [10], and subsequently it has shown significant resistance to almost every insecticide applied in the field, including new chemical compounds [11,12]. In addition, diamondback moth has the distinction of being the first insects to develop resistance in the field to the bacterial insecticide *Bacillus thuringiensis* [13,14]. The resistance of *P. xylostella* populations to *B. thuringiensis* has been observed by [15-23] in

the USA (Florida, Hawaii, and New York), Central America (Mexico, Costa Rica, Guatemala, Honduras, and Nicaragua), and Asia (Japan, China, Malaysia, and the Philippines). In Brazil, [24] it was documented this pest's resistance in environments where *B. thuringiensis* is commonly used as a bioinsecticide.

This has prompted increased efforts worldwide to develop IPM programs for *P. xylostella*, based principally on new management tactics that are not yet used in the field for this pest [8,25,26]. In this chapter, we give an overview of the association of *P. xylostella* with its host plants and natural enemies, and describe management strategies and practices for control of the diamondback moth.

2. Tactics for integrated pest management

2.1. Biological control

Biological control can be defined as the use of one type of organism to reduce the population density of another. Biological control has been used for approximately two millennia, and has been widely used in pest management since the end of the nineteenth century [27]. The following types of biological control can be distinguished: natural, conservative, inoculative (or classical), and augmentative. Natural biological control involves the reduction of pest organisms by their natural enemies and has been occurring since the evolution of the first terrestrial ecosystems, 500 million of years ago [28]. It takes place in all of the world's ecosystems without any human intervention, and, in economic terms, is the greatest contribution of biological control to agriculture [29]. Conservation biological control consists of human actions that protect and stimulate the performance of naturally occurring enemies [30]. In inoculative biological control, natural enemies are collected in an exploration area (usually the area of origin of the pest) and then released in new areas where the pest was accidentally introduced. In augmentative biological natural control, natural enemies are mass-reared in biofactories for release in large numbers to obtain immediate pest control [28].

2.1.1. Entomophagous agents: parasitoids and predators

Parasitoids can be defined as insects that are only parasitic in their immature stages, kill their host in the process of development, and have free-living adults that do not move their hosts to nests or hideouts [31].

All stages of the diamondback moth are attacked by numerous parasitoids and predators, with parasitoids being the more widely studied. Over 90 parasitoid species attack the diamondback moth [32]. Egg parasitoids belonging to the polyphagous genera *Trichogramma* and *Trichogrammatoidea* contribute little to natural control and require frequent mass releases. Larval parasitoids are the most predominant and effective. Many of the effective larval parasitoids belong to two major genera, *Diadegma* and *Cotesia*; a few *Diadromus* spp., most of which are pupal parasitoids, also exercise significant control [1]. The majority of these spe-

http://dx.doi.org/10.5772/54110

cies come from Europe where the diamondback moth is believed to have originated [1]. In countries near Brazil, such as Argentina, *P. xylostella* larval parasitoids collected in the field include the species *Diadegma insulare* (Cresson) (Hymenoptera: Ichneumonidae), *Oomyzus sokolowskii* (Kurdjumov) (Hymenoptera: Eulophidae), and *C. plutellae* (Kurdjumov) (Hymenoptera: Braconidae) [33].

Seven species of parasitoids were observed in a *P. xylostella* population on cabbage crops in the Brasilia region of Brazil, with the two most common species being *Diadegma liontiniae* (Brethes) (Hymenoptera: Ichneumonidae) and *Apanteles piceotrichosus* (Blanchard) (Hymenoptera: Braconidae). *Cotesia plutellae* (Kurdjumov) (Hymenoptera: Braconidae) and *Actia* sp., previously more abundant, had become very minor parasitoids. Six species of hyperparasitoids emerged from *D. liontiniae* and *A. piceotrichosus*, showing a high diversity of natural enemies in this region of recent colonization by *P. xylostella* [34].

In organically farmed kale in Pernambuco, Brazil, seven natural enemies of *P. xylostella* were observed: three parasitoids, *C. plutellae* Kurdjumov (Hymenoptera: Braconidae), *Conura pseudofulvovariegata* (Becker) (Hymenoptera: Chalcididae) and *Tetrastichus howardi* (Olliff) (Hymenoptera: Eulophidae), and four predators, *Cheiracanthium inclusum* (Hentz) (Araneae: Miturgidae), *Pheidole* sp. Westwood (Hymenoptera: Formicidae), and nymphs and adults of *Podisus nigrispinus* (Dallas) (Hemiptera: Pentatomidae) [35].

Several studies have been conducted in Brazil to examine whether these entomophagous agents of the diamondback moth could be used as a biological control for this pest in crucifer crops.

Parasitoids of the genus *Trichogramma* are among the entomophagous agents that have already been studied for *P. xylostella*. The species *T. pretiosum* Riley (Hymenoptera: Trichogrammatidae), Tp8 strain, can parasitize approximately 15 *P. xylostella* eggs in the first or second generation when reared in this host under laboratory conditions, with 100% emergence, and 10 to 11 days for adult emergence [36]. Eggs of two *P. xylostella* populations, one reared on kale leaves and the other on broccoli leaves, were exposed to the *T. pretiosum* Tp8 strain, and the number of parasitized eggs was 5.8–9.4 on kale and 3.2–8.4 on broccoli [37]. Furthermore, the optimal way to mass rear this parasitoid in the laboratory is to use eggs glued to blue, green, or white colored cards [37].

The impact on non-target species, particularly *Trichogramma*, of insecticides for *P. xylostella* control should be analyzed because some are toxic to these parasitoids in crucifers. Endosulfan and etofenprox, classified as class-4 toxic products, are extremely toxic to the parasitoids. Triflumuron, classified as a non-toxic product, is selective for these parasitoids in the eggs of *P. xylostella* [26]. The combination of chemicals or natural insecticidal products from vegetables with certain cultivars of crucifers enables more effective management of the diamondback moth, particularly in the case of the interaction between pyroligneous extract and cabbage. However, the interaction among cultivars and products can be detrimental to the effectiveness of *T. pretiosum* and *T. exiguum*, and thus requires a careful evaluation to minimize the impact on these natural enemies [38]. Bioinsecticides based on *B. thuringiensis* for controlling *P. xylostella* can influence the parasitoid *T. pretiosum* in the moth's eggs. The ap-

plication of isolates of *B. thuringiensis* on *P. xylostella* larvae influenced the parasitism of *T. pretiosum* in eggs of subsequent pest generations [39].

Another parasitoid of *P. xylostella* larvae, which has been studied in Brazil, is *O. sokolowskii*. The duration of the immature stage of these parasitoids can range from 12.9 to 31.6 days at 28 and 18°C, respectively, and the number of adults emerged per pupa of *P. xylostella* varies between 7.3 and 12, with a sex ratio of between 0.86 and 0.91 [40]. During a year, the number of generations of *O. sokolowskii* is always higher than that of *P. xylostella*, suggesting that *O. sokolowskii* could develop up to 24 generations per year while the diamondback moth could reach 20 annual generations [40]. Furthermore, the *O. sokolowskii* parasitoid is able to disperse and parasitize *P. xylostella* throughout a kale field up to 24 meters from the release point [41].

Another larval parasitoid studied in Brazil for *P. xylostella* is *A. piceotrichosus*, which was collected in the Rio Grande do Sul State. Its immature stage was observed to last 14.6 to 15.5 days and its adult longevity was found to be 12.7 to 13.4 days [42].

Among the stink bug predators, *P. nigrispinus* has great potential for use in *P. xylostella* control. *P. nigrispinus* has been reported preying on *P. xylostella* in crucifer crops [35], and, furthermore, this predator consumed on average 10.9 larvae or 5.5 pupae in 24 h [43]. Adults of *Orius insidiosus* (Say) (Hemiptera: Anthocoridae) has been reported consuming 5.9 diamondback moth eggs in 24 h [44].

2.1.2. Entomopathogens: Bacteria

The occurrence of *P. xylostella* populations of resistant to certain active ingredients, like synthetic and biological insecticides, has caused a considerable increase in research directed at developing tactics for Integrated Pest Control based on economic, social, and ecological parameters [21,45-47).

Recent studies on control strategies and population reduction of *P. xylostella* using microorganisms has been increasingly cited in the scientific community, with emphasis on the entomopathogenic bacterium *B. thuringiensis* Berliner (1911) [48-51,39].

This entomopathogen can be easily found in different environments [52,53], and it is characterized by a variety of strains, each forming one or more protein crystals (Cry) and cytolytic toxins [54] that have insecticidal activity and determined its efficiency as a control on certain agricultural pests. Another type of insecticidal protein that can be synthesized by some strains of *B. thuringiensis* is "Vegetative Insecticidal Proteins" (Vip), whose insecticide action spectrum operates in different insect species [55].

A long history of intensive research has established that their toxic effect is due primarily to their ability to form pores in the plasma membrane of the midgut epithelial cells of susceptible insects [56,57]. The presently available information still supports the notion that *B. thuringiensis* Cry toxins act by forming pores, but most events leading to their formation, following binding of the activated toxins to their receptors, remain relatively poorly understood [58].

http://dx.doi.org/10.5772/54110

Strains of *B. thuringiensis* can produce from one to five toxins that represent a large variability in toxicity and interfere in the expression levels and the spectrum control of insects, and differ in their specificity to certain species [59]. For example, the Cry proteins are show high toxicity to insects of the orders Lepidoptera, Coleoptera, Hymenoptera, Diptera, Orthoptera, and Mallophaga, and to other organisms such as nematodes and mites [60,54,61].

Among the different protein crystals identified in insect control, 59 toxins were tested against 71 Lepidoptera species [62]. The broadest range of toxins was tested against *P. xylostella* (43 toxin types), which was one of only 12 species that were tested against 15 toxins or more [62].

In Brazil, *P. xylostella* is controlled using entomopathogenic bacteria in phytosanitary applications of formulation products properly registered for a particular crop, most commonly biological products containing *B. thuringiensis* var. *kurstaki*, which expresses Cry1Aa, Cry1Ab, and Cry1Ac toxins [49] (Table 1).

Crops	Commercial Products	Chemical Groups	Active Ingredients	Formulation	Class	
					Toxicological	Environmental
Broccoli	Bac Control	Biological	*B. thuringiensis* subsp. *kurstaki*	WP	IV	IV
	Dipel	Biological	*B. thuringiensis* subsp. *kurstaki*	WP	II	IV
	Thuricide	Biological	*B. thuringiensis* subsp. *kurstaki*	WP	IV	IV
Cauliflower	Bac Control	Biological	*B. thuringiensis* subsp. *kurstaki*	WP	IV	IV
	Dipel	Biological	*B. thuringiensis* subsp. *kurstaki*	WP	II	IV
	Thuricide	Biological	*B. thuringiensis* subsp. *kurstaki*	WP	IV	IV
Cabbage	Able	Biological	*B. thuringiensis* subsp. *kurstaki*	SC	III	IV
	Agree	Biological	*B. thuringiensis* subsp. *aizawai* GC 91 + *B. thuringiensis* subsp. *kurstaki*	WP	III	IV
	Bac Control	Biological	*B. thuringiensis* subsp. *kurstaki*	WP	IV	IV
	Dipel	Biological	*B. thuringiensis* subsp. *kurstaki*	WG	II	IV
	Dipel	Biological	*B. thuringiensis* subsp. *kurstaki*	WP	II	IV
	Thuricide	Biological	*B. thuringiensis* subsp. *kurstaki*	WP	IV	IV
	Xentari	Biological	*B. thuringiensis* subsp. *aizawai*	WG	II	III
Kale	Bac Control	Biological	*B. thuringiensis* subsp. *kurstaki*	WP	IV	IV
	Dipel	Biological	*B. thuringiensis* subsp. *kurstaki*	WP	II	IV
	Thuricide	Biological	*B. thuringiensis* subsp. *kurstaki*	WP	IV	IV

Source: [63]. WP = Wettable Powder; WG = Water-Dispersible Granules; SC = Suspension concentrate.

Table 1. Commercial products based on *Bacillus thuringiensis* recommended for controlling the population of *Plutella xylostella* in different brassica crops.

However, the low variability in the number of toxins related to formulated biological products, combined with a high number of applications in the field, puts selection pressure on the population of *P. xylostella* and, consequently, expression of resistance of this pest to protein crystals has been observed since the 1990s [20,24].

The development of resistance in *P. xylostella* populations is related to the binding of these toxins with the intestinal epithelium, which occurs through the same membrane receptors [19,22].

Some alternative methods of resistance management of this pest towards *B. thuringiensis* toxins can reduce resistance and even make it possible to break the resistance to biological products [22,64].

According to [49], mixed formulations of different bacteria or isolates of *B. thuringiensis* that have a wide variety of *Cry* toxins, organized in isolation or together, have the ability to reduce selection pressure and, consequently, the development of new cases of resistance in populations of *P. xylostella*.

To improve the biological control of *P. xylostella* using this entomopathogenic bacterium, several studies have initially focused to on the characterization of new strains of *B. thuringiensis*, with the objective of discovering more efficient insecticides and implementing them in new formulations [65,66,51]. In a study conducted by [49] using stored grains and different strains of *B. thuringiensis* from soils of several regions of Brazil, there was high mortality (98–100%) of second-instar larvae of *P. xylostella*. These results have demonstrated that a high variability of *Cry* genes in the same strain can constitute a substantial tool for resistance management of this pest, with subsequent use in the synthesis of new biological products.

In pathogenicity tests, the strains behave in different ways, and few of them are able to cause total mortality in the insects analyzed. In research conducted by [51], approximately 19% of the strains tested caused total mortality to second-instar larvae of *P. xylostella* between 24 and 48 hours.

In this case, in addition to pathogenicity and virulence tests, researchers should analyze the sublethal effects of these strains on the remaining individuals, an important parameter in the toxicological evaluation of *B. thuringiensis* strains [67,68].

Many biological characteristics of *P. xylostella* may be influenced by the sublethal effects of these toxins, causing discernible changes in insect behavior, such as appetite loss, decreased movement with subsequent paralysis, change in the tegument color from bright green to dark yellow or dark brown, and loss of reaction to touch [69,51].

According to [51] and [8], the most pronounced biological changes observed between phytosanitary applications with strains and commercial products based on *B. thuringiensis* were in the viability of larvae and pupae and the weight of pupae. The biological characteristics less influenced by these strains were related to the caterpillar and pupal period and sex ratio [8].

The behavior of strains or commercial products based on *B. thuringiensis* that result in individuals surviving phytosanitary application, but that provide sublethal effects in subsequent generations, may be a significant tool for Integrated Pest Management [8], the objective of which is to improve management of the pest through interactions with other control methods, such as biological control with predators and parasitoids, which will reduce the population density due to sublethal effects caused by strains of *B. thuringiensis*. The remaining pests may be a food source and host for other insects considered beneficial to agriculture, and can help maintain and assist the populations of these arthropods in different crops.

The Integrated Management of *P. xylostella* based on biological control with the entomopathogenic bacterium *B. thuringiensis* is an important method for reducing the population den-

http://dx.doi.org/10.5772/54110

sity of this pest in brassica crops. However, the use of this control must be well planned, because there are populations of this pest resistant to biological products, necessitating the use of certain methods of resistance management to eliminate these harmful individuals and, perhaps, prevent future problems with the development of resistant populations that can undermine the whole program of rational control of this pest.

2.1.3. Entomopathogens: Fungi

There is no fungus-based bioinsecticide registered for crucifer crops in Brazil; however, some entomopathogenic fungi have been studied to determine their potential as a biological control agent for *P. xylostella*. Among the fungi that have been studied for their activity against *P. xylostella*, *Paecilomyces tenuipes* caused the highest mortality to third-instar *P. xylostella* larvae, with an LC_{50} of 1.09×10^6 spores/mL at 25°C [70].

The most active crude protein extract, isolated from the CNZH strain of *Isaria fumosorosea*, produced 83.3% mortality in third-instar larvae 6 days post treatment [71]. Furthermore, it has been found that a synergism exists between the fungus *I. fumosorosea* and a plant secondary chemical, and that larval deaths were directly related to the concentration of each component in the mixtures and their cumulative effect was evident for an extended period [72].

In addition to the species already mentioned, the fungi *Metarhizium anisopliae* and *Beauveria bassiana* were also studied for the control of *P. xylostella*. Several strains from Benin were tested and found to cause 94% larval mortality [73]. Strains obtained in Brazil have also been tested and caused mortality to *P. xylostella* larvae ranging from 70% to 96% [74].

2.1.4. Entomopathogens: Nematodes

Research on the control of Lepidoptera with entomopathogenic nematodes has focused on the diamondback moth [75]. Field studies on cabbage in Java (Indonesia) confirmed that *Steinernema carpocapsae* can be used as a substitute for ineffective chemical insecticides [76]. Diamondback moth eggs are deposited and the emerging larvae feed on the underside of the leaves. The control of young caterpillars with entomopathogenic nematodes can therefore be optimized by directing the nematode spray to the lower side of the leaves [75]. The use of a surfactant for lowering the surface tension and of a polymer for increasing the viscosity significantly improved nematode performance against *P. xylostella* [77]. The performance of these adjuvants is, however, influenced by the spray application technique [75].

2.2. Chemical control

The chemical control method, recommended as one of the tools or tactics of Integrated Pest Management, is still the main strategy for reducing pest populations among crucifer producers. This preference is due to the practicality, speed, and efficiency of controlling insects considered agricultural pests, particularly *P. xylostella* [78].

The chemical groups used to control this pest have great variability in terms of the active ingredient, formulation, and toxicological and environmental classes (Table 2).

Crops	Chemical Group	Active Ingredient	Formulation	Class	
				Toxicological	Environmental
Broccoli	Pyrethroid	Deltamethrin	EC	III	I
	Oxime Methylcarbamate	Methomyl	SL	I	II
	Organophosphate	Acephate	SP	II	III
Canola	Pyrethroid	Bifenthrin	EC	II	II
Cauliflower	Pyrethroid	Deltamethrin	EC	III	I
	Pyrethroid	Permethrin	EC	III	II
	Organophosphate	Acephate	SP	II	III
	Naphthyl Methylcarbamate	Carbaryl	WP	III	II
Cabbage	Anthranilamide+Pyrethroid	Clorantraniliprole+Lambda-cyhalothrin	SC	II	I
	Tetranortriterpenoid	Azadirachtin	EC	III	IV
	Benzoylurea	Teflubenzuron	SC	IV	II
	Benzoylurea	Lufenuron	EC	IV	II
	Benzoylurea	Novalurom	EC	III	II
	Pyrethroid	Deltamethrin	EC	III	I
	Pyrethroid	Permethrin	EC	I	II
	Benzofuranil Methylcarbamate	Carbofuran	GR	III	II
	Oxime Methylcarbamate	Methomyl	SL	I	II
	Organophosphate	Acephate	SP	II	III
	Analog of Pyrazol	Chlorfenapyr	SC	III	II
	Phenilthiourea	Diafenthiuron	WP	I	II
	Anthranilamide	Chlorantraniliprole	SC	III	II
	Oxadiazine	Indoxacarb	WG	I	III
	Naphthyl Methylcarbamate	Carbaryl	WP	III	II
	Spinosyns	Spinosad	SC	IV	III
Kale	Pyrethroid	Deltamethrin	EC	III	I
	Oxime Methylcarbamate	Methomyl	SL	I	II
	Organophosphate	Acephate	SP	II	III
	Pyrethroid	Permethrin	EC	III	II

Source: [63]. EC = Emulsion Concentrate; SL = Soluble Concentrate; SP = Soluble Powder; WP = Wettable Powder; SC = Suspension concentrate; GR = Granules; WG = Water-Dispersible Granules.

Table 2. Chemical groups and active ingredients registered for *Plutella xylostella* control in different brassica crops.

Among the pesticides recommended for different brassicas, the chemical group of pyrethroids represents one of the most important for *P. xylostella* control. Chemical control of *P. xylostella* using a synthetic pyrethroid is recommended when larval density exceeds an economic threshold, which varies in relation to the growth stage of the crop and environmental conditions [79,80]. However, the inappropriate use of these chemical products has considerably increased the frequency of resistance in different diamondback moth populations to some types of active ingredients of this chemical group [81,82,24,83]. According to [84] and [82], *P. xylostella* populations are considered very prone to developing resistance to some active ingredients. In addition to lowering the pesticide efficiency, increasing the frequency of application may not lead to a significant reduction in crop damage.

This may be due to the biological characteristics of this species, the life cycle of which is short when compared to that of other insects, and to the cultural practice of constantly applying pesticides with the same active ingredients in more concentrated doses, without providing a chemical molecule rotation or an appropriate dosage as listed on the label of the phytosanitary product used [24].

http://dx.doi.org/10.5772/54110

In the context of Integrated Pest Management, cultural, physical, plant resistance, biological, and chemical control methods may be important strategies in the success of the *P. xylostella* control program [85]. Techniques such as crop residue removal, management of the interval between crops, use of tolerant cultivars, use of sprinkler irrigation, application of plant and biological products and reduction in the number of pesticide applications by measuring the economic injury level, used harmoniously and consciously, can provide significant improvement in the quality of products and the system in which the culture is embedded [86-90,83].

After a rational application of chemical controls, the first response observed in the field is the high larval mortality of *P. xylostella* in direct proportion to the commercial product concentration recommended for the determined culture [91,83]. Another response to phytosanitary application is a significant alteration in the life cycle of the insect, principally the larval period, because many chemical compounds present in insecticides affect the process of ecdysis, interfering with the transition between instars, and thereby act as a growth regulator [83].

Among the types of insecticides recommended for the control of *P. xylostella*, growth regulators have been found to have low interference with the activity of predators, parasitoids, and entomopathogenic fungi, because they do not affect the embryogenesis and reproduction of this pest, which is important since the parasitoid larvae live inside the pest's eggs before emerging as adults [85,90,38]. This is important principally because the physiological selectivity of this chemical group makes them more toxic to the pest than to the biological control agent [92,93,38,94,26].

Insecticides of plant origin are also a very important group for the population management of this pest. Among these, neem extract (*Azadirarachta indica*) has shown significant results in the control of *P. xylostella*, affecting the growth, larval mortality during ecdysis, oviposition, deformation in pupae and adults, and the physiological processes of reproduction, such as inadequate egg maturation and infertility, that interfere with larval hatching [95,90,83,38,96].

In this context, managing the population of *P. xylostella* using chemical control methods can be a very interesting strategy if well used, because of the large number of chemical groups with different active ingredients, which enables a chemical molecule rotation and prevents the development of resistance. These products can be used with other control techniques to reduce the number of applications of pesticide and improve the quality of the final product. Another very important consideration in choosing the chemical product is its selectivity, because many chemicals have high selectivity for the host but not for biological control agents, which contributes to the maintenance of populations considered beneficial to the integrated management of *P. xylostella*.

2.3. Plant resistance

The crop forms a template for various interactions between pests and their environment, and varietal resistance to pests is a key component for stabilizing an IPM system [97].

Plants have a bewildering array of responses to herbivory, broadly categorized as direct and indirect defenses and tolerance [98]. Some primary wax components, including specific

long-chain alkyl components, have allelochemical activity that influences the host acceptance behavior of *P. xylostella* larvae [99]. Furthermore, glucosinolates, a category of secondary products, are found primarily in species of the Brassicaceae. When tissue is damaged, for example by herbivory, glucosinolates are degraded in a reaction catalyzed by thioglucosidases, called myrosinases, which are also present in these species. This causes the release of toxic compounds such as nitriles, isothiocyanates, epithionitriles and thiocyanates. The glucosinolate-myrosinase system is generally believed to be part of the plant's defense against insects, and possibly also against pathogens [100].

Among various cultivars of crucifers observed, the cabbage 'chato de quintal' showed a high level of the substance glucobrassicin, and was classified as moderately resistant to *P. xylostella* [101].

Several studies have been conducted in Brazil to determine the crucifer cultivars resistant to *P. xylostella* for use in the management of this pest. Among the crucifers that are marketed in Brazil—cabbage cultivars, broccoli, kale, and cauliflower—cabbage cultivars were more resistant, and kale cultivars were more susceptible to diamondback moth [8]. When compared only cultivars of kale, it was found that 'Ribeirão Pires I-2620' was the most susceptible to two generations of diamondback moth [102].

The use of silicon in the integrated management of diamondback moths may help to reduce the use of pesticides. Silicon damages the jaws of larvae, limiting ingestion and causing high mortality [103].

2.4. Cultural control

The current pest management tactics pursued by growers focus on the protection of crucifer seedlings, using both cultural and chemical means, in some seasons in the established crops [104]. Because of the failure of insecticides to control the diamondback moth, interest is growing in the use of cultural controls in commercial crucifer production. Some of the classical control measures that have been tried with some success are intercropping, use of sprinkler irrigation, trap cropping, crop cover rotation, and clean cultivation [1].

The mortality of *P. xylostella* was significantly higher with the intercropping of Chinese cabbage (*Brassica chinensis*) with garlic (*Allium sativum*) and lettuce (*Lactuca sativa*) than in monocultures of Chinese cabbage. These results suggest that intercropping can suppress the diamondback moth populations for a long period rather than just the short term [105]. Furthermore, studies conducted in Brazil of the intercropping of cabbages with other crop plants (cabbage and green onion, cabbage and cilantro, and cabbage, green onion, and cilantro) did not reduce the rate of parasitism of *P. xylostella* larvae by *O. sokolowskii*, which makes it promising for diamondback moth biological control; however it did not interfere with cabbage colonization by the diamondback moth [41].

A study investigating the impact of irrigation systems on diamondback moth infestation in cabbage noted that when irrigation water was applied by sprinkler-irrigation, diamondback moth infestations were reduced by 37.5–63.9% compared with a drip-irrigated control [106].

Glucosinolates are biologically active natural products characteristic of crucifers, and crucifer-specialist insect herbivores, such as *P. xylostella*, frequently use glucosinolates as oviposition stimuli. Benzylglucosinolate-producing tobacco plants were more attractive for oviposition by female *P. xylostella* than wild-type tobacco plants. As newly hatched *P. xylostella* larvae were unable to survive on tobacco, these results represent a proof-of-concept strategy for rendering non-host plants attractive for oviposition by specialist herbivores with the long-term goal of generating efficient dead-end trap crops for agriculturally important pests [107].

With regard to crop cover for crucifers, a broccoli cover-cropping system (cereal rye) resulted in fewer leaves, smaller plants, and a slightly reduced yield when compared to the other systems. Strip-cropping broccoli with potatoes did not convey any agronomic advantages. Gross margin analysis revealed that on a total system basis, a 2.2% yield improvement or a 7% price premium was required to make the cover crop system perform as well as conventional practice [108].

Another study looked at the effect of two diversification strategies, one a broccoli/potato (*Solanum tuberosum*) strip crop comprising 1.65-m (tractor width) replications of two rows of potatoes and two rows of broccoli, and the other a cereal rye (*Secale cereale*) cover crop, which formed a sacrificial planting that was killed and rolled flat to minimize weed competition and improve the agronomic performance of the subsequent broccoli crop. In this case, it was observed that *P. xylostella* eggs, and the subsequent larvae and pupae, were less abundant on broccoli with the cover crop, probably due to interference with host location and oviposition processes. The strip crop had no effect on broccoli crop yield [109].

2.5. Sex pheromones

The potential for using synthetic sex pheromone traps as a simple and practical method of monitoring population densities of insect pests has been investigated in many crop systems. Sex pheromones of *P. xylostella* have already been synthesized for use in the management of this pest in crucifers [110]. Thus, trap catches can be used to forecast infestations during periods that coincide with high *P. xylostella* infestations [111].

Currently, pheromone-baited traps in the Prairie Pest Monitoring Network are used to detect and survey [112] the arrival of migrating moths. Recent research has shown that capture of male moths in pheromone-baited traps in the Prairie Pest Monitoring Network is correlated with moderate, but not low, densities of the immature stages of the diamondback moth sampled in the same fields [113]. Then exists the potential to develop commercially available pheromone-baited traps as tools that can predict the ephemeral nature of diamondback moth population densities in the prairies and inform producers of key thresholds and timing for control efforts [113].

When placed on Delta sticky traps, the artificial sex pheromone Bioplutella, marketed in Brazil, efficiently captured males of the diamondback moth and could be used for monitoring this pest [114].

3. Final remarks

As shown above, the management of pests on crucifers in Brazil has largely been dependent on synthetic pesticides, used prophylactically or in response to *P. xylostella* occurrence, although cultural practices have also played some role in the control of the diamondback moth [104]. The general lack of understanding of interactions between crucifers and their invertebrate pests and between pests and their natural enemies has resulted in a lack of alternative integrated options for growers. Growers would rely less heavily on the prophylactic and reactive application of broad-spectrum pesticides if they were provided with knowledge and training in identifying natural enemies and using economic thresholds. Furthermore, we again emphasize glucosinolates, their breakdown products, and plant volatile compounds as key components in these processes [115], which have been considered beneficial in the past and hold great promise for the future of integrated pest management.

Author details

S.A. De Bortoli, R.A. Polanczyk, A.M. Vacari, C.P. De Bortoli and R.T. Duarte

Department of Plant Protection, FCAV-UNESP, Jaboticabal, Sao Paulo, Brazil

References

[1] Talekar NS, Shelton AM. Biology, ecology, and management of the diamondback moth. Annual Review of Entomology 1993;38(1): 275–301.

[2] Sarfraz M, Dosdall LM, Keddie BA. Diamondback moth-host plant interactions: implications for pest management. Crop Protection 2006;25(7): 625–639.

[3] Hardy JE. *Plutella maculipennis* Curt., its natural and biological control in England. Bulletin of Entomological Research 1938;29(4): 343–372.

[4] Kfir, R. Origin of the diamondback moth (Lepidoptera: Plutellidae). Annals of the Entomological Society of America 1998;91(2): 164–167.

[5] Liu S, Wang X, Guo S, He J, Shi Z. Seasonal abundance of the parasitoid complex associated with the diamondback moth, *Plutella xylostella* (Lepidoptera: Plutellidae) in Hangzhou, China. Bulletin of Entomological Research 2000;90(3): 221–231.

[6] Shelton AM. Management of the diamondback moth: déjà vu all over again? In: Endersby NM, Ridland PM. (eds.) Proceedings of the fourth international workshop on the management on the diamondback moth and other crucifer pests. Melbourne, Victoria, Australia; 2001. p3–8.

[7] Castelo Branco M, França FH, Villas Boas GL. Traça-das-crucíferas (*Plutella xylostella*). Brasília: Embrapa Hortaliças; 1997. 4p.

[8] De Bortoli SA, Vacari AM, Goulart RM, Santos RF, Volpe HXL, Ferraudo AS. Capacidade reprodutiva e preferência da traça-das-crucíferas para diferentes brassicáceas. Horticultura Brasileira 2011;29(2): 187–192.

[9] Smith DB, Sears MK. Evidence for dispersal of diamondback moth, *Plutella xylostella* (L.) (Lepidoptera: Plutellidae), into southern Ontario. Proceedings of the Entomological society of Ontario, Canada. 1982;113: 21–27.

[10] Ankersmit GW. DDT resistance in *Plutella maculipennis* (Curt.) Lepidoptera in Java. Bulletin of Entomological Research 1953;44: 421–425.

[11] Sarfraz M, Keddie BA. Conserving the efficacy of insecticides against *Plutella xylostella* (L.) (Lepidoptera: Plutellidae). Journal of Applied Entomology 2005;129(3): 149–157.

[12] Ridland PM, Endersby NM. Some Australian populations of diamondback moth, *Plutella xylostella* (L.) show reduced susceptibility to fipronil. In: Srinivasan R, Shelton AM, Collins HL. (eds.) Sixth international workshop on management of the diamondback moth and other crucifer insect pests. Nakhon Pathom, Thailand; 2011. p21–25.

[13] Kirsch K, Schmutlerer H. Low efficacy of a *Bacillus thuringiensis* (Berl.) formulation in controlling the diamondback moth *Plutella xylostella* (L.), in the Philippines. Journal of Applied Entomology 1988;105(1-5): 249–255.

[14] Tabashnik BE, Cushing NL, Finson N, Johnson MW. Field development of resistance to *Bacillus thuringiensis* in diamondback moth (Lepidoptera: Plutellidae). Journal of Economic Entomology 1990;83(5): 1671–1676.

[15] Gong Y, Wang C, Yang Y, Wu S, Wu Y. Characterization of resistance to *Bacillus thuringiensis* toxin Cry1Ac in *Plutella xylostella* from China. Journal of Invertebrate Pathology 2010;104(2): 90–96.

[16] Sayyed AH, Gatsi R, Palacios SI, Escriche B, Wright DJ, Crickmore N. Common, but complex, mode of resistance of *Plutella xylostella* to *Bacillus thuringiensis* toxins Cry1Ab and Cry1Ac. Applied of Environmental Microbiology 2005;71(11): 6863–6869.

[17] Zhao JZ, Collins HL, Tang JD, Cao J, Earle ED, Roush RT, Herrero S, Escriche B, Ferré J, Shelton AM. Development and characterization of diamond back moth resistance to transgenic broccoli expressing high levels to Cry1C. Applied and Environmental Microbiology 2000;66(9): 3784–3789.

[18] Díaz-Gomez O, Rodríguez JC, Shelton AM, Lagunes A, Bujanos R. Susceptibility of *Plutella xylostella* (L.) (Lepidoptera: Plutellidae) populations in Mexico to commercial formulations of *Bacillus thuringiensis*. Journal of Economic Entomology 2000;93(3): 963–970.

[19] Tang JD, Shelton AM, Van Rie J, De Roeck S, Moar WJ, Roush RT, Peferoen M. Toxicity of *Bacillus thuringiensis* spore and crystal protein to resistant diamondback moth (*Plutella xylostella*). Applied and Environmental Microbiology 1996;62(2): 564–569.

[20] Tabashnik BE. Evolution of resistance to *Bacillus thuringiensis*. Annual Review of Entomology 1994;39(1) 47–79.

[21] Perez CJ, Shelton AM. Resistence of *Plutella xylostella* (Lepidoptera: Plutellidae) to *Bacillus thuringiensis* Berliner in Central America. Journal of Economic Entomology 1997;90(1): 87–93.

[22] Wright DJ, Iqbal M, Granero F, Ferre J. A change in a single midgut receptor in the diamondback moth (*Plutella xylostella*) is only part responsible for field resistance to *Bacillus thuringiensis* subsp. *kurstaki* and *Bacillus thuringiensis* subsp. *aizawai*. Applied and Environmental Microbiology 1997;63(5): 1814–1819.

[23] Ferré J, Real MD, van Rie J, Jansens S, Peferoen M. Resistance to the *Bacillus thuringiensis* bioinsecticide in the field population of *Plutella xylostella* is due to a change in a midgut membrane receptor. Proceeding of the National Academic of Science 1991;88(12): 5119–5123.

[24] Castelo Branco M, França FH, Pontes LA, Amaral PST. Avaliação da suscetibilidade a inseticidas em populações da traça-das-crucíferas de algumas áreas do Brasil. Horticultura Brasileira 2003;21(3): 549–552.

[25] Sarfraz M, Dosdall LM, Keddie BA. Influence of the herbivore host's wild food plants on parasitism, survival and development of the parasitoid *Diadegma insulare*. Biological Control 2012;62(1): 38–44.

[26] Goulart RM, Volpe HXL, Vacari AM, Thuler RT, De Bortoli SA. Insecticide selectivity to two species of *Trichogramma* in three different hosts, as determined by IOBC/WPRS methodology. Pest Management Science 2012;68(2): 240–244.

[27] van Lenteren J, Godfray HCJ. Europen in science in the Enlightenment and the discovery of the insect parasitoid life cycle in The Netherlands and Great Britain. Biological Control 2005;32(1): 12–24.

[28] van Lenteren, J. The state of commercial augmentative biological control: plenty of natural enemies, but a frustrating lack of uptake. BioControl 2012;57(1): 1–20.

[29] Waage JK, Greathead DJ. Biological Control: challenges and opportunities. Philosophical Transactions of the Royal Society of London 1988;318(1189): 111–128.

[30] Gurr GM, Wratten SD. Measures of success in biological control. Dordrecht: Kluwer Academic Publishers; 2000, p430.

[31] Godfray HCJ. Parasitoids: behavioural and evolutionary ecology. New Jersey: Princeton University Press; 1994, p488.

[32] Goodwin S. Changes in the numbers in the parasitoid complex associated with the diamondback moth, *Plutella xylostella* (L.) (Lepidoptera) in Victoria. Australian Journal of Zoology 1979;27(6): 981–989.

[33] Bertolaccini I, Sánchez DE, Arregui MC, Favoro JC, Theiler N. Mortality of *Plutella xylostella* (Lepidoptera, Plutellidae) by parasitoids in the Province of Santa Fe, Argentina. Revista Brasileira de Entomologia 2011;55(3): 454–456.

[34] Guilloux T, Monnerat R, Castelo-Branco M, Kirk A, Bordat D. Population dynamics of *Plutella xylostella* (Lep., Yponomeutidae) and its parasitoids in the region of Brasilia. Journal of Applied Entomology 2003;127(5): 288–292.

[35] Silva-Torres CSA, Pontes IVAF, Torres JB, Barros R. New records of natural enemies of *Plutella xylostella* (L.) (Lepidoptera: Plutellidae) in Pernambuco, Brazil. Neotropical Entomology 2010;39(5): 835–838.

[36] Volpe HXL, De Bortoli AS, Thuler RT, Viana CLTP, Goulart RM. Avaliação de características biológicas de *Trichogramma pretiosum* Riley (Hymenoptera: Trichogrammatidae) criado em três hospedeiros. Arquivos do Instituto Biológico 2006;73(3): 311–315.

[37] Magalhães GO, Goulart RM, Vacari AM, De Bortoli SA Parasitismo de *Trichogramma pretiosum* Riley, 1879 (Hymenoptera: Trichogrammatidae) em diferentes hospedeiros e cores de cartelas. Arquivos do Instituto Biológico 2012;79(1): 55–90.

[38] Thuler RT, De Bortoli SA, Goulart RM, Viana CLTP, Pratissoli D. Interação tritrófica e influência de produtos químicos e vegetais no complexo: brássicas x traça-das-crucíferas x parasitóides de ovos. Ciência e Agrotecnologia 2008;32(4): 1154–1160.

[39] De Bortoli AS, Vacari AM, Magalhães GO, Dibelli W, De Bortoli CP, Alves MP. Subdosagens de *Bacillus thuringiensis* em *Plutella xylostella* (Lepidoptera: Plutellidae) e *Trichogramma pretiosum* (Hymenoptera: Trichogrammatidae). Revista Caatinga 2012;25(2) 50–57.

[40] Ferreira SWJ, Barros R, Torres JB. Exigências térmicas e estimava do número de gerações de *Oomysus sokolowshii* (Kurdjumov) (Hymenoptera: Eulophidae), para regiões produtoras de crucíferas em Pernambuco. Neotropical Entomology 2003;32(3): 407–411.

[41] Silva-Torres CSA, Torres JB, Barros R. Can cruciferous grown under variable conditions influence biological control of *Plutella xylostella* (Lepidoptera: Plutellidae)? Biocontrol Science and Technology 2011;21(6): 625–641.

[42] Gonçalves RR, Di Mare RA. Biologia da traça das crucíferas, *Plutella xylostella* Linnaeus (Lepidoptera: Yponomeutidae), sob condições controladas de temperatura, e parasitoides assiciados. Parte III. Estudo sobre a biologia de *Apanteles piceotrichosus* (Blanchard) (Hymenoptera: Braconidae): análise do efeito de endocruzamento. Revista Brasileira de Zoologia 2005;22(3): 806–809.

[43] Vacari AM, De Bortoli SA, Torres JB. Relation between predation by *Podisus nigrispinus* and developmental phase and density of its prey, *Plutella xylostella*. Entomologia Experimentalis et Applicata 2012;145(1): 30–37.

[44] Brito JP, Vacari AM, Thuler RT, De Bortoli SA. Aspectos biológicos de *Orius insidiosus* (Say, 1832) predando ovos de *Plutella xylostella* (L., 1758) e *Anagasta kuehniella* (Zeller, 1879). Arquivos do Instituto Biológico 2009;76(4): 627–633.

[45] Kogan, M. Integrated pest management: historical perspectives and contemporary development. Annual Review of Entomology 1998;43: 243–270.

[46] Baek JH, Kim JL, Lee D, Chung BK, Miyata T, Lee SH. Identification and characterization of ace1-type acetylcholinesterase likely associated with organophosphate resistance in *Plutella xylostella*. Pesticide Biochemistry and Physiology 2005;81(3): 164–175.

[47] Medeiros PT, Sone EH, Soares CMS, Dias JMCS, Monnerat RG. Avaliação de produtos à base de *Bacillus thuringiensis* no controle da traça-das-crucíferas. Horticultura Brasileira 2006;24(2): 245–248.

[48] Khan MFR, Griffin RP, Carner GR, Gorsuch CS. Susceptibility of Diamondback Moth, *Plutella xylostella* (L.) (Lepidoptera: Plutellidae), from collard fields in South Carolina to *Bacillus thuringiensis*. Journal of Agricultural and Urban Entomology 2005;22(1): 19–26.

[49] Thuler AMG, Thuler RT, Cícero ES, De Bortoli SA, Lemos MVF. Estudo da variabilidade gênica em isolados brasileiros de *Bacillus thuringiensis* para emprego no controle biológico de *Plutella xylostella*. Boletín de Sanidad Vegetal Plagas 2007;33(3): 409–417.

[50] Baxter SW, Zhao J, Shelton AM, Vogel H, Heckel DG. Genetic mapping of Bt-toxin binding proteins in a Cry1A-toxin resistant strain of diamondback moth *Plutella xylostella*. Insect Biochemistry and Molecular Biology 2008;38(2): 125–135.

[51] Viana CLTP, De Bortoli SA, Thuler RT, Goulart RM, Thuler AMG, Lemos MVF, Ferraudo AS. Efeito de novos isolados de *Bacillus thuringiensis* Berliner em *Plutella xylostella* (Linnaeus, 1758) (Lepidoptera: Plutellidae). Científica 2009;37(1): 22–31.

[52] Maeda M, Mizuki E, Nakamura Y, Hatano T, Ohba M. Recovery of *Bacillus thuringiensis* from marine sediments of Japan. Current Microbiology 2000;40(6): 418–422.

[53] Martínez C, Caballero P. Contents of cry genes and insecticidal activity of *Bacillus thuringiensis* strains from terrestrial and aquatic habitats. Journal of Applied Microbiology 2002;92(4): 745–752.

[54] BRAVO, A.; GILLL, S. S.; SOBERÓN, M. Mode of action of *Bacillus thuringiensis* Cry and Cyt toxins and their potential for insect control. Toxicon 2007;49(4): 423–435.

[55] Estruch JJ, Warren GW, Mullins MA, NYE GJ, Craig JA. Vip3A, a novel *Bacillus thuringiensis* vegetative insecticidal protein with a wide spectrum of activities against

Lepidopteran insects. Proceedings of the National Academy of Science 1996;93(11): 5389–5394.

[56] Knowles BH, Ellar DJ. Colloid-osmotic lysis is a general feature of the mechanism of action of *Bacillus thuringiensis* δ-endotoxins with different insect specificity. Biochimica et Biophysica Acta 1987;924(3): 509–518.

[57] Kurouac M, Vachon V, Noel JF, Girard F, Schwartz JL, Laprade R. Amino acid and divalent ion permeability of the pores formed by the *Bacillus thuringiensis* toxins Cry1Aa and Cry1Ac in insect midgut brush border membrane vesicles. Biochimica et Biophysica Acta 2002;1591(2): 171–179.

[58] Vachon V, Laprade R, Schwartz JL. Current models of the mode of action of *Bacillus thuringiensis* insecticidal crystal proteins: a critical review. Journal of Invertebrate Pathology 2012;111(1): 1–12.

[59] Mohan M, Sushil SN, Selvakumar G, Bhatt JC, Gujar GT, Gupta HS. Differential toxicity of *Bacillus thuringiensis* strains and their crystal toxins against high-altitude Himalayan populations of diamondback moth, *Plutella xylostella* L. Pest Management Science 2009;65(1): 27–33.

[60] Crickmore N, Zeigler DR, Feitelson J, Schnepf E. van Rie J, Lereclus D, Baum J, Dean DH. Revision of the nomenclature for the *Bacillus thuringiensis* pesticidal crystal proteins. Microbiology and Molecular Biology Reviews 1998;62(3): 807–813.

[61] Silveira LFV, Polanczyk RA, Pratissoli D, Franco CR. Seleção de isolados de *Bacillus thuringiensis* Berliner para *Tetranychus urticae* Koch. Arquivos do Instituto Biológico 2011;78(2): 273-278.

[62] van Frankenhuyzen K. Insecticidal activity of *Bacillus thuringiensis* crystal proteins. Journal of Invertebrate Pathology 2009;101(1): 1–16.

[63] Ministério da Agricultura Pecuária e Abastecimento. Agrofit: sistema de agrotóxicos fitossanitários. http://agrofit.agricultura.gov.br/agrofit_cons/principal_agrofit_cons/ (accessed 15 August 2012).

[64] Maxwell EM, Fadamiro HY, Mclaughlin JR. Suppression of *Plutella xylostella* and *Trichoplusia ni* in cole crops with attracticide formulations. Journal of Economic Entomology 2006;99(4): 6–17.

[65] Monnerat RG, Leal-Bertioli SCM, Bertioli DJ, Butt TM, Bordat D. Caracterização de populações geograficamente distintas da traça-das-crucíferas por suscetibilidade ao *Bacillus thuringiensis* Berliner e RAPD-PCR. Horticultura Brasileira 2004;22(3): 607–609.

[66] Medeiros PT, Pereira MN, Martins ES, Gomes ACMM, Falcão R, Dias JMCS, Monnerat RG. Seleção e caracterização de estirpes de *Bacillus thuringiensis* efetivas no controle da traça-das-crucíferas *Plutella xylostella*. Pesquisa Agropecuária Brasileira 2005;40(11):1145–1148.

[67] Salama HS, Foda MS, El-Sharaby A, Matter M, Khalafallah M. Development of some lepidopterus cotton pests as affected by exposure to sublethal levels of endotoxins of *Bacillus thuringiensis* for different periods. Journal of Invertebrate Pathology 1981;38(2): 220–229.

[68] Polanczyk RA, Alves S. *Bacillus thuringiensis*: uma breve revisão. Agrociência 2003;7(2): 1–10.

[69] Silva LKF, Carvalho AG. Patogenicidade de *Bacillus thuringiensis* (Berliner, 1909) em lagartas de *Urbanus acawoios* (Williams, 1926) (Lepidoptera: Hesperiidae). Arquivos do Instituto Biológico 2004;71(2): 249–252.

[70] Baksh A, Khan A. Pathogenicity of *Paecilomyces tenuipes* to diamondback moth, *Plutella xylostella* at three temperatures in Trinidad. International Journal of Agriculture and Biology 2012;14(2): 261–265.

[71] Freed S, Jin FL, Naeem M, Ren SX, Hussian M. Toxicity of proteins secreted by Entomopathogenic fungi against *Plutella xylostella* (Lepidoptera: Plutellidae). International Journal of Agriculture and Biology 2012;14(2): 291–295.

[72] Xu D, Ali S, Huang Z. Insecticidal activity influence of 20-Hydroxyecdysone on the pathogenicity of *Isaria fumosorosea* against *Plutella xylostella*. Biological Control 2011;56(3): 239–244.

[73] Godonou I, James B, Atcha-Ahowé C, Vodouhe S, Kooyman C, Ahanchédé A, Korie S. Potential of *Beauveria bassiana* and *Metarhizium anisopliae* isolates from Benin to control *Plutella xylostella* (L.) (Lepidoptera: Plutellidae). Biological Control 2009;28(3): 220–224.

[74] Silva VCA, Barros R, Marques EJ, Torres JB. Suscetibilidade de *Plutella xylostella* (L.) (Lepidoptera: Plutellidae) aos fungos *Beauveria bassiana* (Bals.) Vuill. e *Metarhizium anisopliae* (Metsch.) Sorok. Neotropical Entomology 2003;32(4): 653–658.

[75] Brusselman E, Beck B, Pollet S, Temmerman F, Spanoghe P, Moens M, Nuyttens D. Effect of the spray application technique on the deposition of entomopathogenic nematodes in vegetables. Pest Management Science 2012;68(3): 444–453.

[76] Schroer S, Sulistyanto D, Ehlers RU. Control of *Plutella xylostella* using polymer-fomulated *Steinernema carpocapsae* and *Bacillus thuringiensis* in cabbage fields. Journal of Applied Entomology 2005;129(4): 198–204.

[77] Schroer S, Ziermann D, Ehlers RU. Mode of action of a surfactant-polymer formulation to support performance of the entomopathogenic nematode *Steinernema carpocapsae* for control of diamondback moth larvae (*Plutella xylostella*). Biocontrol Science and Technology 2005;15(6): 601–613.

[78] Castelo Branco M, Melo CA. Resistência a abamectin e cartap em populações de traça-das-crucíferas. Horticultura Brasileira 2002;20(4): 541–543.

[79] Miles M. Insect Pest Management II – *Etiella*, False Wireworm and Diamondback Moth. GRDC Research updates. http://www.grdc.com.au, 2002 (accessed 20 August 2012).

[80] Micic S. Chemical Control of Insect and Allied Pests of Canola. Farmnote No. 1/2005. Department of Agriculture, South Perth, Western Australia, Australia; 2005.

[81] Carazo ER, Cartin VML, Monge AV, Lobo JAS, Araya LR. Resistencia de *Plutella xylostella* a deltametrina, metamidofós y cartap em Costa Rica. Manejo Integrado de Plagas 1999;53: 52–57.

[82] Castelo Branco M, França FH, Medeiros MA, Leal JGT. Uso de inseticidas para o controle da traça-do-tomateiro e da traça-das-crucíferas: um estudo de caso. Horticultura Brasileira 2001;19(1): 60–63.

[83] Thuler RT, De Bortoli SA, Barbosa JC. Eficácia de inseticidas químicos e produtos vegetais visando ao controle de *Plutella xylostella*. Científica 2007;35(2): 166–174.

[84] Georghiou G, Lagunes-Tejada A. The ocurrence of resistance to pesticides in arthropods. An index of cases reported through 1989. Rome: FAO; 1991. p318.

[85] Gallo D, Nakano O, Silveira Neto S, Carvalho RPL, Baptista GC, Berti Filho E, Parra JRP, Zucchi RA, Alves SB, Vendramim JD, Marchini LC, Lopes JRS, Omoto C. Entomologia Agrícola. Piracicaba: FEALQ; 2002. p920.

[86] Carballo VM, Hruska AJ. Períodos críticos de proteccion y efecto de la infestacion de *Plutella xylostella* L. (Lepidoptera: Plutellidae) sobre el rendimiento del repollo. Manejo Integrado de Plagas 1989;14: 46–60.

[87] Castelo Branco M, Villas Bôas GL, França FH. Nível de dano de traça-das-crucíferas em repolho. Horticultura Brasileira 1996;14(2): 154–157.

[88] Castelo Branco M, Gatehouse AG. Insecticide resistance in *Plutella xylostella* (Lepidoptera: Yponomeutidae) in the Federal District, Brazil. Anais da Sociedade Entomológica do Brasil 1997;26(1): 75–79.

[89] Ulmer BC, Gillot C, Woods D, Erlandson M. Diamondback moth, *Plutella xylostella* (L.), feeding and oviposition preferences on glossy and waxy *Brassica rapa* (L.) lines. Crop Protection 2002;21(4): 327–331.

[90] De Bortoli SA, Thuler RT, Lopes BS. Efeito de lufenuron e azadiractina sobre adultos de *Plutella xylostella* (Lepidoptera: Plutellidae). Científica 2006;34(1): 53–58.

[91] Lima MPL, Barros R. Toxicidade de lufenuron para lagartas de *Plutella xylostella* (L., 1758) (Lepidoptera: Plutellidae). Revista Ômega 2000;9(1): 52–54.

[92] O'Brien RD. Toxic phosphorus esters. New York: Academic; 1960. p434.

[93] Czepak C, Fernandes PM, Santana HG, Takatsuka FS, Rocha CL. Eficiência de inseticidas para o controle de *Plutella xylostella* (Lepidoptera: Plutellidae) na cultura do re-

polho (*Brassica oleraceae* var. *capitata*). Pesquisa Agropecuária Tropical 2005;35(2): 129–131.

[94] Bacci L, Picanço MC, Da Silva EM, Martins JC, Chediak M, Sena ME. Seletividade fisiológica de inseticidas aos inimigos naturais de *Plutella xylostella* (L.) (Lepidoptera: Plutellidae) em brássicas. Ciência e Agrotecnologia 2009;33: 2045–2051.

[95] Mordue AJ, Blackwell A. Azadirachtin: an update. Journal of Insect Physiology 1993;39(11): 903–924.

[96] Jesus FG, Paiva LA, Gonçalves VC, Marques MA, Boiça Júnior AL. Efeito de plantas inseticidas no comportamento e biologia de *Plutella xylostella* (Lepidoptera: Plutellidae). Arquivos do Instituto Biológico 2011;78(2): 279–285.

[97] Panda N, Khush GS. Host Plant Resistance to Insects. Wallingford, USA: CAB International; 1995, p448.

[98] Kessler A, Baldwin IT. Plant responses to insect herbivore: the emerging molecular analysis. Annual Review of Plant Biology 2002;53: 299–328.

[99] Eigenbrode SD, Pillai SK. Neonate *Plutella xylostella* responses to *surfasse* wax componentes of a resistant cabbage (*Brassica oleraceae*). Journal of Chemical Ecology 1998;24(10): 1611–1627.

[100] Rask L, Andréasson E, Ekbom B, Eriksson S, Pontoppidan B, Meijer J. Myrosinase: gene family evolution and herbivore defense in Brassicaceae. Plant Molecular Biology 2000;42(1): 93–113.

[101] Thuler RT, De Bortoli SA, Hoffmann-Campo CB. Classificação de cultivares de brássicas com relação a resistência a traça-das-crucíferas e a presença de glucosinolatos. Pesquisa Agropecuária Brasileira 2007;42(4): 467–474.

[102] Boiça Junior AL, Tagliari SRA, Pitta RM, Jesus FG, Braz LT. Influência de genótipos de couve (*Brassica oleracea* L. var. *acephala* DC.) na biologia de *Plutella xylostella* (L., 1758) (Lepidoptera: Plutellidae). Ciência e Agrotecnologia 2011;35(4): 710–717.

[103] Freitas LM, Junqueira AMR, Michereff Filho M. Potencial de uso do silício no manejo integrado da traça-das-crucíferas, *Plutella xylostella*, em plantas de repolho. Revista Caatinga 2012;25(1): 8–13.

[104] Gu H, Fitt GP, Baker GH. Invertebrate pests of canola and their management in Australia: a review. Australian Journal of Entomology 2007;46(3): 231–243.

[105] Cai HJ, Li SY, Ryall K, You MS, Lin S. Effects of intercropping of garlic or lettuce with Chinese cabbage on the development of larvae and pupae of diamondback moth (*Plutella xylostella*). African Journal of Agricultural Research 2011;6(15): 3609–3615.

[106] Mchugh JJ, Foster RE. Reduction of diamondback moth (Lepidoptera: Plutellidae) infestation in head cabbage by overhead irrigation. Journal of Economic Entomology 1995;88(1): 162–168.

[107] Moldrup ME, Geu-Flores F, De Vos M, Olsen CE, Sun J, Jander G, Halkier BA. Engineering of benzylglucosinolate in tobacco provides proof-of-concept for dead-end trap crops genetically modified to attract *Plutella xylostella* (diamondback moth). Plant Biotechnology Journal 2012;10(4): 435–442.

[108] Broad ST, Lisson SN, Mendham NJ. Agronomic and gross margin analysis of an insect pest suppressive broccoli cropping system. Agricultural Systems 2009;102(1–3): 41–47.

[109] Broad ST, Schellhorn NA, Lisson SN, Mendham NJ. Host location and oviposition of lepidopteran herbivores in diversified broccoli cropping systems. Agricultural and Forest Entomology 2008;10(2): 157–165.

[110] Zong GH, Yan SQ, Liang XM, Wang DQ, Zhang JJ. Synthesis of the sex pheromone of *Plutella xylostella* (L.). Chinese Journal of Organic Chemistry 2011;31(12): 2126–2130.

[111] Nofemela RS. The ability of synthetic sex pheromone traps to forecast *Plutella xylostella* infestations depends on survival of immature stages. Entomologia Experimentalis et Applicata 2010;136(3): 281–289.

[112] Hopkinson RF, Soroka JJ. Air trajectory model applied to an in-depth diagnosis of potential diamondback moth infestations on the Canadial Prairies. Agricultural and Forest Meteorology 2010;150(1): 1–11.

[113] Evenden ML, Gries R. Assessment of commercially available pheromone lures for monitoring diamondback moth (Lepidoptera: Plutellidae) in Canola. Journal of Economic Entomology 2010;103(3): 654–661.

[114] Imenes SDL, Campos TB, Rodrigues Netto SM, Bergmann EC. Avaliação da atratividade de feromônio sexual sintético da traça-das-crucíferas, *Plutella xylostella* (L.) (Lepidoptera: Plutellidae), em cultivo orgânico de repolho. Arquivos do Instituto Biológico 2002;69(1): 81–84.

[115] Ahuja I, Rohloff J, Bones AM. Defence mechanisms of Brassicaceae: implications for plant-insect interactions and potential for integrated pest management. A review. Agronomy for Sustainable Development 2010;30(2): 311–348.

IntechOpen