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CHAPTER - 8

Integrated Pest Management in Tropical Vegetable Crops

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8.1 Introduction

Vegetables are cultivated over an area of about 19 million ha globally, with an annual production of 268 million tonnes; Asia and Africa account for more than 90% of the hectarage and production (FAO, 2011). Vegetable crops contribute to human and environmental health. Most vegetables can supply plant proteins, vitamins (especially vitamins A and C), folic acid, minerals such as iron and calcium, and dietary fiber to the human diet (Fowler, 2011). Vegetable legumes also fix atmospheric nitrogen in the soil, thus improving soil fertility. Their haulms can be used as high quality livestock fodder. Most vegetables are high-value, repeat-cycle crops that can help lift small-scale farmers out of poverty in developing tropical countries (Genova *et al.*, 2010). However, vegetable production is often constrained by several severe biotic- and abiotic factors that reduce yields and profits.

Insect pests and diseases are one of the major production constraints in tropical vegetable production. For example, the eggplant fruit and shoot borer (EFSB), *Leucinodes orbonalis* Guenée (Lepidoptera: Pyralidae) can reduce yield by as much as 90% (Parimi and Zehr, 2009). Up to 80% yield losses have been reported in tomato due to damage by its fruit borer, *Helicoverpa armigera* Hubner (Lepidotera: Noctuidae) (Tewari and Krishnamoorthy, 1984). Tomato yellow leaf curl virus disease transmitted by the whitefly, *Bemisia tabaci* (Gennadius) (Hemiptera: Aleyrodidae) can reach up to 100% incidence with yield losses often exceeding 90% (Saikia and Muniyappa, 1989). A loss of 30 to 100% can be caused in various cucurbits by the melon fly, *Bactrocera cucurbitae* (Coquillett) (Diptera: Tephritidae) (Dhillon *et al.*, 2005). As a major pest of vegetable brassicas, diamondback moth, *Plutella xylostella* (Linnaeus) (Lepidoptera: Plutellidae) causes up to 50% yield losses (Sandur, 2004). Significant yield losses of up to 50% have been attributed to onion thrips, *Thrips tabaci* Lindeman (Thysanoptera: Thripidae) in bulb alliums (Mote, 1976).

Owing to the significant loss of marketable yield due to pests and diseases in the tropics, farmers tend to apply extremely high quantities of chemical pesticides to vegetable crops, often in an indiscriminating fashion. For example, farmers may spray pesticides more than 50 times on their tomato crops in a given season in India (Nagaraju *et al.*, 2002). Cambodian farmers spray up to 20 times per season using up to five different pesticides on major vegetable crops (Sodavy *et al.*, 2000). Cabbage producers in Benin applied approximately 46.8 l/ha of pesticide concentrate on the crop using 19 applications (Rosendahl *et al.*, 2008). Weekly spraying of pesticides in vegetable production is routine in Cameroon (Abang *et al.*, 2013).

With few vegetable pesticides available in sub-Saharan Africa, farmers apply pesticides registered for other crops, often in incorrect dosages; some may even use banned pesticides (Williamson, 2003; Amoah *et al.*, 2006). Pesticide misuse and/or overuse are thus not uncommon in the tropics, and lead to the development of resistance in target insects. Pesticides are often applied to vegetables as the time for harvest approaches in many countries (Middleton *et al.*, 2004; Hoi, 2010), which leaves residue on the fruit that cannot be reduced to levels suitable for human consumption (Neufeld *et al.*, 2010). The adverse effects of these harmful pesticides on the health of consumers are considered to be high (Brink *et al.*, 2003).

It is likely that integrated pest management (IPM) strategies can achieve a sufficient level of pest control to be acceptable to vegetable producers. Various IPM strategies have been developed and are being promoted among farmers in the tropics. This chapter aims to compile simple, affordable, and environmentally sound IPM strategies, developed mostly by AVRDC – The World Vegetable Center and/or its collaborators, for the management of key pests in selected tropical vegetable crops.

8.2 Eggplant, *Solanum melongena* L. (Solanales: Solanaceae)

The major pests of eggplant include the fruit and shoot borer (*L. orbonalis*), leafhopper [*Amrasca devastans* Distant (Hemiptera: Cicadellidae)], whitefly

(*B. tabaci*), thrips [*Thrips palmi* Karny (Thysanoptera: Thripidae)] and aphid [*Aphis gossypii* Glover (Hemiptera: Aphididae)].

The sucking pests such as whitefly, aphid, thrips and leafhopper first appear on the crop either in the seedling nursery or in the main field soon after transplanting. Depending on management practices, they may be present in the crop throughout the cropping season. Both the adults and nymphs of *B. tabaci* suck the plant sap and reduce the vigor of the plant. In severe infestations, the leaves turn yellow and drop off. Whiteflies secrete large quantities of honeydew, which favors the growth of sooty mould on leaf surfaces and reduces the photosynthetic efficiency of the plants. *A. gossypii* occurs in large numbers on the tender shoots and lower leaf surfaces, and sucks the plant sap. Slightly infested leaves exhibit yellowing. Severe aphid infestations cause young leaves to curl and become deformed. Like whitefly, aphids also produce honeydew, which leads to the development of sooty mould.

Both nymphs and adults of *A. devastans* suck the sap from the leaves, which leads to yellowing, bronzing and drying, or “hopper burn”. *T. palmi* prefers to feed mostly on foliage, but occasionally on fruit. Slightly infested leaves exhibit silvery feeding scars on the lower leaf surfaces, especially along the mid-rib and veins. In severe infestations, the leaves turn yellow or brown and dry on the lower leaf surfaces. Infested fruit is scarred and deformed.

During the early vegetative stage of the crop, the *L. orbonalis* larva feeds inside the tender shoots, which results in wilting of the young shoots, followed by drying and drop-off. The pest prefers to feed on the fruit during the fruiting stage of the crop; damaged fruit also exhibits boreholes on the surface that often are sealed with excreta, rendering the fruit unfit for marketing and consumption.

Integrated Pest Management

8.2.1. Crop rotation with non-solanaceous crops is an effective way to reduce the population build-up of *L. orbonalis*. However, careful consideration in choosing the crops for rotation is imperative, because the crops selected (*e.g.,* cotton or okra) may also support other common pests such as whitefly, leafhopper and aphid.

8.2.2. Planting resistant or moderately resistant cultivars can reduce pest damage and thus allow greater yields. For example, accessions or varieties such as EG058, Pusa Purple Long, Pusa Purple Cluster, Pusa Purple Round, etc. have been reported to be tolerant or resistant to *L. orbonalis* (Parker *et al.*, 1995; Alam *et al.*, 2003; Shivalingaswamy and Satpathy, 2007). Except for EG058, which is an AVRDC accession, most of the varieties noted originated in India. Indian cultivars such as 'Manjari Gota,' 'Vaishali,' 'Mukta Kesi,' 'Round Green,' and 'Kalyanipur T3' and the Bangladeshi variety, Bagun 6, are reported to be less susceptible or tolerant to *A. devastans* damage (Parker *et al.*, 1995; Rashid *et al.*, 2003). Three cultivated eggplant accessions (S130, S145 and S158) are reported to be less susceptible to *A. devastans* and *A. gossypii* (AVRDC, 1999). However, no reported resistant or tolerant eggplant varieties are available for *B. tabaci* and *T. palmi*.

8.2.3. Eggplant seedlings should not be grown near fields with previous or existing crops, or near dried eggplant heaps (Alam *et al.*, 2003). If seedlings must be grown in such areas, the seedling beds should be covered with 30-mesh nylon net to prevent the entry of *L. orbonalis* moths and thus egg laying. If *B. tabaci* and *T. palmi* are common in the region, use of 50–64 mesh nylon net to cover the seedling beds is useful preventive measure. If non insect-proof nets are used, they should be treated with insecticides or neem oil to prevent the entry of sucking insects into the protective structures (Cerkauskas, 2004).

8.2.4. Yellow (565-590 nm) sticky traps can be used to monitor and/or mass-trap *B. tabaci* and *A. gossypii* in the field (Durairaj *et al*., 2007). Similarly, blue (H"460-475 nm) sticky traps can be used to monitor and/or mass-trap *T. palmi* (Chen *et al.*, 2004; Naojee *et al.*, 2010).

8.2.5. Prompt removal and destruction of infested shoots and fruit at regular intervals until final harvest throughout a community substantially reduces *L. orbonalis* damage (Alam *et al.*, 2003). Pruning does not adversely affect the plant growth or yield (Srinivasan and Huang, 2009).

8.2.6. *L. orbonalis* sex pheromone lures in traps can be installed at the rate of 100 traps per hectare (Cork *et al.*, 2003) to reduce fruit damage and increase yield. Traps should be placed either at canopy level or at slightly above the canopy level for effective attraction (Alam *et al.*, 2003).

8.2.7. Generalist predators such as ladybird beetles and green lacewings are highly efficient in preying on sucking pests. Parasitoids such as *Anagrus flaveolus* Waterhouse and *Stethynium triclavatum* Enock are effective against *A. devastans* (Subba Rao 1968; Parker *et al.,* 1995). Parasitoids such as *Trathala flavoorbitalis* (Cameron), *Eriborus sinicus* Holmgren, and *Pristomerus testaceus* Morley are commonly found occurring on *L. orbonalis* larvae (AVRDC, 1996a;

Alam *et al.*, 2003). In addition, weekly releases of the egg parasitoid *Trichogramma chilonis* Ishii @ 1 g parasitized eggs/ha/week, and larval parasitoid *Bracon hebetor* Say @ 800-1000 adults/ha/week are effective in reducing *L. orbonalis* (Alam *et al.*, 2006). Broad-spectrum pesticides should not be used, as they may harm these and other natural enemies.

8.2.8. Microbial pesticides and botanical pesticides can be used to manage *L. orbonalis* and other sucking pests (Rao *et al.*, 1999; Islam *et al.*, 2011; Mathur *et al.*, 2012). Need based application of appropriately selective pesticides, when recommended by the local extension service, is also advisable.

8.3 Tomato, *Solanum lycopersicum* L. (Solanales: Solanaceae)

Several kinds of insect and mite pests attack tomato leaves, flower buds, and fruit during tomato cultivation. The most commonly occurring pests on tomato are fruit worm (*H. armigera*), common armyworm *[Spodoptera litura* Fabricius (Lepidoptera: Noctuidae)], whitefly (*B. tabaci*), leaf miner [*Liriomyza* spp. (Diptera: Agromyzidae)] and spider mites [*Tetranychus urticae* Koch*.,* *T. cinnabarinus* (Boisd.) and *T. evansi* Baker & Pritchard (Acarina: Tetranychidae)]. Whitefly and fruit worm are the major insect pests in most of the tropical tomato-producing regions of the world, as the whitefly transmits leaf-curl virus disease and fruit worm causes severe damage to the fruit, reducing the marketable yield of tomato.

Liriomyza spp. will appear on the crop in the seedling stage as well as in the main field soon after transplanting. Their larval feeding causes irregular mines on leaf surfaces. In severe infestations, several mines are formed on the same leaf, which will drastically reduce photosynthesis and thus reduce yield. Sometimes, death of the whole plant occurs. Although *B. tabaci* feeding can cause direct damage to the growing plants, it also acts as a vector for tomato yellow leaf-curl virus (TYLCV) disease. Plants infected by TYLCV show stunted growth with erect shoots. Leaflets curl upwards and inwards, are reduced in size, are stiff, thicker than normal, and leathery in texture. Leaves have yellowing along the margins and have interveinal chlorosis (yellowing). The flowers wither and droop, and fruit set is reduced or may be nil. Fruit, if produced, is small and unmarketable.

The neonate larvae of *S. litura* feed on leaf surfaces. Mature larvae feed on the whole leaves, leaving only the main veins. Rarely, they feed on the immature growth stages of tomato fruit. However, *S. litura* does not bore into fruit like *H. armigera.* Sometimes, the larvae may cut the seedlings or young plants at soil level. *H. armigera* is a polyphagous and highly mobile insect. The older larvae prefer to feed on flowers and young fruits. The larvae make holes and then feed by thrusting their heads inside. The holes are circular and often surrounded by fecal pellets.

Spider mites have emerged recently as serious pests of tomato in South Asia, Southeast Asia, and Africa. Low relative humidity favors their multiplication and hence they pose a serious production threat during the dry season. Spider mite feeding leads to the formation of several white or yellow speckles on the leaves. In severe infestations, leaves will completely desiccate and drop-off. The mites also produce webbing on the leaf surfaces; under severe conditions, the whole plant is wrapped in webs. Under high population densities, the mites move to the tip of the leaf or top of the plant and congregate to form a ball-like mass, which will be carried by the wind to newer leaves or plants.

Integrated Pest Management

8.3.1. Crop rotation with non-host crops is effective in managing tomato pests. However, selection of an appropriate non-host crop is challenging, because most of the tomato pests are highly polyphagous and feed on several agricultural and horticultural crops as well as weed plants.

8.3.2. Although using resistant tomato cultivars can reduce pest damage, commercial cultivars with appreciable levels of resistance to major tomato pests are not yet available. Germplasm screening at AVRDC – The World Vegetable Center revealed the presence of high levels of *H. armigera* resistance only in wild *Solanum* species, particularly *S. habrochaites* and *S. pennellii*. Efforts to introgress resistance from wild species into cultivated tomato resulted in small-fruited resistant accessions (Talekar *et al.*, 2006). Although resistant sources, mostly in wild tomatoes, are available for *B. tabaci* (Fancelli and Vendramim, 2002; Toscano *et al.*, 2002; Muigai *et al.*, 2003; Srinivasan and Uthamasamy, 2004; Baldin *et al.*, *2005; Firdaus et al.*, 2012; 2013), cultivation of commercially available TYLCV resistant tomato varieties is currently the suggested practice. For instance, varieties from Southern India such as 'Sankranthi,' 'Nandi,' and 'Vybhav' have been reported to be effectively resistant (Muniyappa *et al.*, 2002).

8.3.3. Healthy seedling production as explained in the previous eggplant section might also prevent or reduce the incidences of early season sucking pests, including whitefly.

8.3.4. African marigold (*Tagetes erecta* L.) can be planted as a trap crop to reduce the incidence of *H. armigera* (Srinivasan *et al.*, 1994). It is important to synchronize transplanting of both crops so that flowering coincides, which attracts *H. armigera* female adults. Tropical soda apple (*Solanum viarum* Dunal) also can be used as an effective trap crop to manage *H. armigera* (AVRDC, 2000; 2001; Srinivasan *et al.*, 2013).

8.3.5. Yellow is the most attractive color to the adults of *Liriomyza* sp. (Parrella, 1987). Hence, yellow sticky traps can be used to monitor and/or mass-trap the adults of *Liriomyza* spp., besides *B. tabaci*.

8.3.6. Sex pheromone traps can be used to monitor, mass-trap, or disrupt the mating of male moths of *H. armigera* and *S. litura*. Although sex pheromone traps baited with pheromone lures can be used to trap more males, this method is less effective for polyphagous insects like *H. armigera* and *S. litura.* Polyphagous populations are always higher due to the availability of multiple host plants in the tropics. Two population parameters *viz.,* density and polymorphism, are important factors that affect the trap catches of males (Kumar and Shivakumara, 2003). Placing a high concentration of sex pheromone (all or one component of the multi-component pheromone) in a slow-release formulation on a 5- and 10-m grid in the field results in a drastic reduction in male moths, which adversely affects mating in *H. armigera* (AVRDC, 1988). Significant trapping of hundreds of male moths has also been achieved in *S. litura* with sex pheromone lures (Dharma Putra *et al.*, 2013; Srinivasan *et al.*, 2013). Hence, mass-trapping is possible when the traps are baited with effective pheromone lures/formulations.

8.3.7. Egg parasitoids (*Trichogramma pretiosum* Riley and *T. chilonis*) and larval parasitoids (*Campoletis chlorideae* Uchida) can be conserved and/or released in tomato fields at regular intervals to check the build-up of *H. armigera* and *S. litura* (Ballal and Singh, 2003; Gupta *et al*., 2004; Krishnamoorthy, 2012). In addition, commercially available biopesticides based on *Bacillus thuringiensis* Berliner, *Helicoverpa armigera* nucleopolyhedrovirus (HaNPV), *Spodoptera litura* nucleopolyhedrovirus (SlNPV), *Beauveria bassiana* (Bals.-Criv.) Vuill. and neem (*Azadirachta indica* A. Juss.) can be used either alone or in combinations against *H. armigera* and *S. litura* (Nathan and Kalaivani, 2006; Mohan *et al.*, 2007; Senthilkumar *et al.*, 2008). Application of *B. bassiana* in the soil has substantially contributed to the mortality of *H. armigera* pupae (AVRDC, 1992). Bio-pesticide formulations based on *B. thuringiensis* subsp. *aizawai* and *B. thuringiensis* subsp. *kurstaki* are highly toxic to the larvae of *H. armigera* (AVRDC, 1995).

8.3.8. Use of broad-spectrum chemical pesticides should be avoided, as these pesticides harm the parasitoid and predator complex of *Liriomyza* spp. and *Tetranychus* spp. Leaf miners have several parasitoids. For instance, *Gronotoma micromorpha* Perkins (larval-pupal parasitoid), *Chrysocharis pentheus* Walker, *Neochrysocharis formosa* (Westwood) and *Diglyphus isaea* Walker (larval parasitoids) and *Halticoptera circulus* Walker and *Opius phaseoli* Fischer (pupal parasitoids) are known to occur in Asia (Lee *et al.*, 1990; Sivapragasam and Syed, 1999; Niranjana *et al.*, 2005; Abe, 2006;), and help to keep the *Liriomyza* spp. population in check. Similarly, several predators of *Tetranychus* spp. occur in most countries. For instance, *Stethorus* spp., *Oligota* spp., *Anthrocnodax occidentalis* Felt, *Feltiella minuta* Felt, etc. are known to occur in Taiwan (Ho, 2000). A Brazilian strain of the predatory mite, *Phytoseiulus longipes* Evans, and the pathogenic fungus, *Neozygites floridana* (Weiser & Muma), have shown promising results against *T. evansi* (Furtado *et al.*, 2007; Wekesa *et al.*, 2007). Oil-based formulations of entomopathogenic fungi such as *B. bassiana* isolate GPK and *Metarhizium anisopliae* (Metchnikoff) Sorokin isolate ICIPE78 also reduced the population density of *T. evansi* substantially (Wekesa *et al.*, 2005). The predatory mite and biopesticides can be used to manage *Tetranychus* spp.

8.3.9. Chemical pesticides are widely used against tomato pests. Pesticide spraying should be scheduled soon after noticing the eggs or during the early larval stages of *H. armigera* and *S. litura*. Selective and systemic pesticides can be used for the sucking insects. It is advisable to follow a proper pesticide rotation. Before application, it is important to check the effectiveness of chemical pesticides in the region and their registration status for tomato.

8.4. Peppers, *Capsicum* spp. (Solanales: Solanaceae)

The major pests on peppers are thrips [*T. palmi*, *Scirtothrips dorsalis* Hood, and *T. parvispinus* Karny (Thysanoptera: Thripidae)], aphids [*A. gossypii* and *Myzus persicae* Sulzer (Hemiptera: Aphididae)], fruit borer (*H. armigera* and *S. litura*), whitefly (*B. tabaci*) and broad mite [*Polyphagotarsonemus latus* (Banks) (Acarina: Tarsonemidae)].

Thrips feed mostly on foliage, especially on the undersides. Slightly infested leaves exhibit silvery to brown feeding scars, especially along the mid-rib and veins. In severe infestations, the leaves curl upward like the shell of a boat, and are reduced in size. Infested fruit are scarred. Thrips also transmit *Tomato spotted wilt virus* and *Capsicum chlorosis virus* (Adkins *et al.*, 2010). Aphids feed on the tender shoots and lower leaf surfaces. The infested leaves are distorted, stunted and curled. The upper leaf surfaces develop sooty mould due to the production of honeydew. Aphids transmit *Chilli veinal mottle virus* and *Cucumber mosaic virus*. Broad mite feeding is usually confined to the undersides of leaves, where areas between veins are brownish and dried out and brittle in severe cases. Young leaves are curled downward and narrower than normal (Parker *et al.*, 1995). Damage by *H. armigera* and *S. litura* during flowering and fruit formation can lead to 90% flower and fruit drop (Reddy and Reddy, 1999).

Integrated Pest Management

8.4.1. Healthy seedling production practices should be followed to reduce the incidence of early season sucking pests.

8.4.2. Selected *Capsicum annuum* L. and *C. baccatum* L. accessions such as PBC018, PBC030, PBC081, PBC151, PBC272, PBC681, PBC726, PBC732, PBC785 and PBC880 were reported to be moderately resistant or resistant to *A. gossypii* (AVRDC, 1993a; 1998). Accessions such as PBC725, PBC901 and PBC1398 were reported to be resistant to *P. latus* (AVRDC, 1998). Accessions such as C00069 and PBC145 were found to be resistant to both *M. persicae* and *T. palmi*. Cultivation of these or similar locally available resistant accessions could substantially reduce incidences of thrips, aphids and broad mite in the tropics.

8.4.3. Castor (*Ricinis communis* L.) can be planted as a trap crop to divert the incidence of *S. litura* on peppers (Reddy and Rosaiah. 1987).

8.4.4. Yellow sticky traps can be used to monitor and/or mass-trap the adults of *B. tabaci*. Blue sticky traps and kairomonal cues baited traps can be used to trap thrips (Nielsen *et al.*, 2010). A semiochemical-baited autoinoculation device treated with *M. anisopliae* could also be adapted to manage thrips on peppers (Niassy *et al.*, 2012).

8.4.5. Sex pheromone traps, biopesticides and parasitoids as described in the tomato section can also be used to manage *H. armigera* and *S. litura* larvae.

8.4.6. Chemical pesticides are widely used against pepper pests. Pesticide spraying should be scheduled soon after noticing eggs or during the early larval stages of *H. armigera* and *S. litura*. A new group of insecticides, the diamides, offer potential control of *H. armigera* and *S. litura* (Tatagar *et al.*, 2009). However, it is advisable to follow a proper pesticide rotation to avoid the development of resistance. A combination of organic compounds containing natural alkaloids, lactone and isoflavones, seaweed extract and chavicol, and neem with avermectins can substantially reduce the leaf curl caused by thrips and broad mite (Mondal and Mondal, 2012).

8.5. Vegetable Brassicas, *Brassica* spp. (Brassicales: Brassicaceae)

Diamondback moth, *P. xylostella,* is a cosmopolitan and destructive pest of vegetable brassicas. The larvae feed and create holes in the leaves; however, severe damage is caused when the larvae tunnel into the heads of crops like cabbage, sometimes causing almost 100% crop loss. Cabbage head caterpillar, *Crocidolomia binotalis* (Lepidoptera: Pyralidae) is another serious pest of brassicas. The early instar larvae migrate toward the growing center of vegetable brassicas, where they conceal themselves in webbing. Subsequent feeding can damage the plant's apical meristem and thus destroy the entire crop (Smyth *et al.*, 2003). Cabbage webworm, *Hellula undalis* (Lepidoptera: Pyralidae) is also a major brassica pest in lowland production systems (Sivapragasam and Chua, 1997), especially in the summer season (AVRDC, 1978). It prefers to feed on the young terminal bud and the unopened leaves (Sivapragasam and Chua, 1997), leading to yield losses of up to 100% in Asia, Africa and the Pacific (Kalbfleisch, 2006). Imported cabbage worm, *Pieris rapae* L. (Lepidoptera: Pieridae) occasionally causes serious damage to vegetable brassicas. The larvae bore directly into the growing heads or cause irregular feeding holes on the leaves. Aphids [*M. persicae*, *Lipaphis erysimi* (Kaltenbach) and *Brevicoryne brassicae* L. (Hemiptera: Aphididae)] also cause significant damage to brassica plants in all growth stages. Aphid infestation leads to leaf curling and wrinkling. With severe infestation, the leaves wilt and the entire plant dies (Parker *et al.*, 1995). The striped flea beetle, *Phyllotreta striolata* Fab. (Coleoptera: Chrysomelidae) causes serious damage to leafy brassicas and radish in Southeast Asia. Infested young plants have numerous small holes in the cotyledons and leaves.

Integrated Pest Management

8.5.1. Growing resistant accessions can avert pest damage. Four non-heading Chinese cabbage accessions (B464, B487, B488 and B490) were found to be tolerant to aphids (AVRDC, 1977). Five Chinese cabbage accessions (B159, B186, B197, B488 and B501) were least susceptible to *H. undalis* (AVRDC, 1978; 1987). Two *Brassica campestris* ssp. *chinensis* var. *parachinensis* accessions (B095 and B583) that are compatible with Chinese cabbage for breeding, were least susceptible to *P. xylostella* (AVRDC, 1981). Cultivation of these, or similar locally available resistant accessions, could substantially reduce the pest incidences on vegetable brassicas in the tropics.

8.5.2. Brassica seedling production can be carried out under 32-mesh nylon net to reduce *P. xylostella* infestation early in the season. Growers can thus reduce the amount of chemical pesticides used at the beginning of the crop cycle, which will help to protect natural enemies. In peri-urban areas where the brassicas are grown on smaller farms, plots can be confined within 32-mesh nylon net barriers on all four sides using a 2-2.5 m high net barrier to manage *P. xylostella* and *C. binotalis* (AVRDC, 1999; 2000; 2001).

8.5.3. Clean cultivation is recommended for *H. undalis* because it feeds and develops on certain weeds such as *Cleome rutidosperma* and *C. viscose*, which act as alternate hosts during the off-season in South and Southeast Asia (Sivapragasam and Chua, 1997; Kalbfleisch, 2006).

8.5.4. Sequential trap cropping with Indian mustard (*Brassica juncea* (L.) Czern.), at the rate of two rows of mustard between every 25 rows of brassica, is an effective way of reducing *P. xylostella* and *C. binotalis* damage. One row of mustard should be sown 15 days before brassica planting, and a second sowing is recommended on the adjacent ridge on the 25th day after planting brassica (Srinivasan and Krishnamoorthy, 1992). The trap crop should be sprayed with chemical pesticides. Yellow rocket, *Barbarea vulgaris* var. *arcuata,* can be used as a dead-end trap crop for *P. xylostella* (Shelton and Badenes-Pérez, 2006). Yellow rocket is highly attractive to *P. xylostella*, but its offspring cannot survive on it. Dead-end trap crops do not require any pesticide treatment to prevent pest populations from moving onto the main crop.

8.5.5. Intercropping of common cabbage with safflower or garlic can substantially reduce the infestation of *P. xylostella* (AVRDC, 1987).

8.5.6. Traps baited with female sex pheromone lures can be placed at the rate of 50 traps per hectare randomly in the field, and replaced twice during the season (AVRDC, 1991) to manage *P. xylostella*. A recent study has shown that sex pheromone traps installed at the rate of 30-45 traps/ha were effective for controlling *P. xylostella* in the field, and reduced insecticide use by 30% per planting season in southwestern China (Zhao *et al.*, 2011). It must be noted that different *P. xylostella* populations may react differently to the same sex pheromone lure (Feng *et al.*, 2011). Hence, the pheromone lures should be validated in a new region before using them as a pest management tool. Traps baited with a 10:1 mixture of (*Z*)-11-hexadecenyl acetate and (*Z*)-9-tetradecenyl acetate attracted substantially high numbers of male *C. binotalis* moths in vegetable brassica fields in Indonesia (Adati *et al.*, 2007). Wing traps baited with female sex pheromone (E,E-11,13-hexadecadienal @ 10 µg per trap) can be placed at a height of 0.5 m above ground at a distance of 15 m, and replaced every two to six weeks depending on the trap catches to manage *H. undalis* (Kalbfleisch, 2006).

8.5.7. Traps baited with aggregation pheromones plus host plant volatiles could trap substantially large numbers of *P. striolata* beetles (Beran *et al.*, 2011; Srinivasan, 2012).

8.5.8. Release of a larval parasitoid (*Diadegma semiclausum* Hellen) and the pupal parasitoid [*Diadromus collaris* (Gravenhorst)] substantially reduced damage from *P. xylostella* in highland areas. The larval parasitoid *Cotesia plutellae* Kurdjumov is well-adapted to manage *P. xylostella* in lowland areas. Unlike the egg parasitoid, augmentative releases for the larval and pupal parasitoids at regular intervals are not necessary. These larval and pupal parasitoids already have been introduced into several countries in Asia (AVRDC, 1993b; 1996b) and Africa (Nyambo and Lohr, 2005). A generalist egg parasitoid, *T. chilonis,* was found to efficiently parasitize egg masses of *C. binotalis* in Samoa (Uelese *et al.*, 2011). Although the polyphagous pentatomid bug, *Eocanthecona furcellata* Wolff, predates more than 70% of the third instar *C. binotalis* larvae, the searching ability in the field decreases (AVRDC, 1996a). However, there is no species-specific efficient parasitoid available for *C. binotalis* hitherto. Release of an egg parasitoid, *Trichogrammatoidea bactrae* Nagaraja once a week for five weeks starting within a week after transplanting brassicas is recommended to manage *H. undalis* (Sivapragasam, 1996). In addition, a few larval parasitoids such as *Bassus* sp., *T. flavoorbitalis*, *Chelonus* sp. and *Phanerotoma* sp. have been reported to infest *H. undalis* in Malaysia. *Apanteles glomeratus* L. is the widely occurring larval parasitoid against *P. rapae*.

8.5.9. *P. xylostella* and *C. binotalis* are susceptible to *B. thuringiensis* toxins, for example, Cry1Ac, Cry1Ab, Cry1Aa, Cry1Ba and Cry1Ca (Srinivasan and Hsu, 2008; Shelton *et al.*, 2009). Hence, *B. thuringiensis* formulations based on the most sensitive Bt toxins would offer effective control (Ooi, 1980; Sastrosiswojo and Setiawati, 1992; Malathi and Sriramulu, 2000). As *B. thuringiensis* biopesticides are not harmful to the parasitoids, both these components synergistically reduce *P. xylostella* and *C. binotalis* damage. Strains of entomopathogenic fungi such as *M. anisopliae* were also effective against *P. xylostella* (AVRDC, 1999). Improved formulations of *B. bassiana* showed promising prolonged impact in suppressing *P. xylostella* populations in field conditions (Ghosh *et al.*, 2011). *H. undalis* is highly sensitive to Cry1Ba and Cry1Ca, but less susceptible to Cry1A toxins (Srinivasan and Hsu, 2008; Shelton et al., 2009). This is also confirmed by its susceptibility to *B. thuringiensis* subsp. *aizawai* based formulations (Srinivasan and Hsu, 2008), but not for the *B. thuringiensis* subsp. *kurstaki* based formulations (AVRDC, 1987), although the latter was shown to be effective in the Philippines (Ulrichs and Mewis, 2003) and India (Singh *et al.*, 2000). Hence, after local validation, *B. thuringiensis* formulations can be used to manage *H. undalis*. Similarly, *P. rapae* is also sensitive to Cry1Ba and Cry1Ca toxins (Shelton *et al.*, 2009), and most formulations of *B. thuringiensis* are effective against this pest. A granulosis virus specifically infecting *P. rapae* is also found in Taiwan (Parker *et al.*, 1995).

8.5.10. Chemical pesticides still occupy the predominant role in managing *P. xylostella*. Due to extensive and inappropriate pesticide use, it has developed resistance to almost every class of insecticide that has been used against it, including novel insecticides such as chlorantraniliprole and flubendiamide (Sukonthabhirom *et al.*, 2011) and biopesticides such as *B. thuringiensis* (Tabashnik *et al.*, 1990). In addition, use of broad-spectrum chemical pesticides early in the season often disrupts the control exerted by parasitoids (Talekar and Shelton, 1993). The rotation and windows approach for pesticide application can effectively manage *P. xylostella* on vegetable brassicas (Baker, 2011; Ridland and Endersby, 2011). Proper selection and judicious use of chemical pesticides targeting *P. xylostella* should also control *C. binotalis* and *H. undalis* on vegetable brassicas; *C. binotalis* is found to be susceptible to newer pesticides such as emamectin benzoate, spinosad and indoxacarb (Kannan *et al.*, 2011). An improved mode of pesticide application via brassica shoots is suggested for *H. undalis* (Sivapragasam, 1996) as this method minimizes the adverse effects of chemical pesticides on the natural enemies present in brassica production systems.

8.6 Cucurbits: *Cucurbita* spp., *Citrullus* spp., *Cucumis* spp., *Momordica* spp. (Cucurbitales: Cucurbitaceae)

Melon fly, *Bactrocera cucurbitae* (Diptera: Tephritidae) has been recorded as a predominant pest on several host plants in the Cucurbitaceae. The extent of losses varies between 30 to 100%, depending on the plant species and the season (Dhillon *et al.*, 2005). Maggots feed inside the fruit, leading to distortion or rotting. The pumpkin caterpillar, *Diaphania indica* Saund. (Lepidoptera: Pyralidae) also causes major yield losses in several cucurbits. It widely occurs in most tropical countries. The young larvae fold the leaves together and feed on the foliage. They also feed on the young fruits, especially on the fruit skin. Aphids (*M. persicae* and *A. gossypii*) also cause significant damage to cucurbits as a direct pest, as well as vectors of viruses such as *Zucchini yellow mosaic virus*, *Watermelon mosaic virus*, *Cucumber mosaic virus* and *Cucurbit aphid-borne yellows virus* (Biswas and Varma, 2006; Bunwaree, 2001; Dahal *et al.*, 1997; Haque *et al.*, 2004; Shaifullah *et al.*, 2005; van der Meer and Garnett, 1987; Wakman *et al.*, 2002). The viruses can cause yield losses of up to 100% (Dahal *et al.*, 1997; Gaba *et al.*, 2004; Provvidenti, 1996). The whitefly (*B. tabaci*) transmits *Squash leaf curl virus* in cucurbits. Symptoms include leaf curling, mottling and severe stunting of the whole plant, and fruit may be small and distorted (Duffus and Stenger, 1998). Up to 100% disease incidence (with 100% yield losses) has been reported (Bananej *et al.*, 2002; Isakeit and Robertson, 1994).

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8.6.1. Sources of resistance to melon fly are available in pumpkin, bitter gourd, bottle gourd, sponge gourd, ridge gourd and round melon (Dhillon *et al.*, 2005; Gogi *et al.*, 2010). However, the resistance may vary across locations. For instance, bitter gourd varieties such as Faisalabad-long and Col-II, reported as the most resistant in Pakistan (Gogi *et al.*, 2010) were highly susceptible to melon fly in Taiwan. Resistance and/or tolerance to *A. gossypii* in *Cucumis melo* L. germplasm has been reported (Bohn *et al.*, 1996; Dhillon *et al.*, 2007; Martin *et al.*, 1996). Resistance in melon accessions to *A. gossypii* has also decreased viral transmission (Lecoq *et al.*, 1979). Aphid resistance has been introduced into several commercial melon cultivars (Lecoq *et al.*, 1998). Three Indian *C. melo* accessions (PI414723, PI164723 and 90625) and a Korean accession (PI161375) had field resistance to *B. tabaci* in some Caribbean islands (Boissot *et al.*, 2003). Thus availability and cultivation of similar locally available resistant accessions could substantially reduce pest incidences on cucurbits.

8.6.2. Healthy seedling production as described in the eggplant section might also prevent or reduce the incidence of early season sucking pests including aphids and whitefly, and also reduce the transmission of viruses in susceptible varieties.

8.6.3. If field sanitation (collection and destruction or burying of infested fruit 15-45 cm deep in the soil) is adopted throughout a community, it will substantially reduce the melon fly population build-up (Klungness *et al.*, 2005; Kumar *et al.*, 2011). Bagging the fruits soon after formation prevents melon fly damage substantially (Akhtaruzzaman *et al.*, 1999; Kumar *et al.*, 2011).

8.6.4. Yellow sticky traps and biopesticides can be used to manage whitefly and aphids.

8.6.5. Kairomonal attractant cue-lure [4-(p-acetoxyphenyl)-2-butanone] traps are highly effective for monitoring and mass-trapping of *B. cucurbitae* in bitter gourd and other crops (Pawar *et al.*, 1991; Vargas *et al.*, 2000). Similarly, protein baits are highly attractive to female melon flies (Kumar *et al.*, 2011). Sex pheromone lures are effective against *D. indica* moths (Wakamura *et al.*, 1998) and they are now commercially available.

8.6.6. An egg – pupal parasitoid, *Fopius arisanus* (Sonan) is believed to have high potential in reducing melon fly damage and it has been introduced into Hawaii and Mexico (Bautista *et al.* 2004; Zenil *et al.*, 2004). *Apanteles taragamae* Viereck, *T. flavoorbitalis* and *Elasmus indicus* Rohwer were found to parasitize *D. indica*, although they are not species-specific parasitoids (Ganehiarachchi, 1997; Muniappan *et al.*, 2012).

8.6.7. Soil application of *M. anisopliae* formulations has reduced the emergence of fruit flies (Ekesi *et al.*, 2005). This strategy can be easily adapted for melon fly. *B. thuringiensis* is effective against *D. indica* (Schreiner, 1991).

8.6.8. Since application of chemical pesticides against melon fly may not be effective, attractants can be combined with small quantities of pesticides to develop an 'attract and kill' system. For example, protein bait sprays mixed with chemical pesticides can be spot-sprayed on the roosting host plants of melon fly. Crops like maize, cassava, sorghum and castor should be planted around the main crop (bitter gourd, water melon, *etc*). These roosting host plants can be spot-sprayed with protein bait sprays once a week (or more often during the wet season) to kill the newly emerged flies before they mature (Vargas *et al.*, 2008; Mcquate, 2011). Since both melon fly and *D. indica* can develop insecticide resistance rapidly (Chen, 2002), chemical pesticides should be selected and applying with care.

8.7 Okra, *Abelmoschus* spp. (Malvales: Malvaceae)

Cotton aphid (*A. gossypii*) and whitefly (*B. tabaci*) are the major pests of okra in its early growth stages. In addition to causing direct feeding damage, *B. tabaci* also transmits yellow vein mosaic virus disease. Early symptoms include alternate green and yellow patches, vein clearing, and vein chlorosis of leaves. The yellow network of veins becomes very conspicuous with thickened veins and veinlets in the later stages. Cotton leafhopper (*A. devastans*) is another major pest of okra. Feeding by the nymphs and adults leads to hopper burn symptoms. Flea beetles [*Podagrica uniformis* (Jac.) and *P. sjostedti* (Jac.) (Coleoptera: Chrysomelidae)] are two serious pests of okra in Africa. Their infestation commences from germination through all stages of plant growth. Although they are mainly leaf eaters (Fasunwon and Banjo, 2010), they also are vectors for *Okra mosaic virus* (Alegbejo *et al.*, 2008). Pod borers [*H. armigera*, *Earias vitella* Fab., *E. insulana* Boisd., *E. biplaga* Walker and *E. cupreoviridis* Walker (Lepidoptera: Noctuidae)] are the major pests of okra during the vegetative as well as fruiting stage. *Earias* spp. bore into the terminal shoots of young plants causing death of the growing tips and leading to the development of lateral branches. During the reproductive stage, the larvae bore and feed into the flower buds, flowers and pods (Varela and Seif, 2004).

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8.7.1. Although sources of resistance to whitefly are not available, varieties that are resistant or moderately resistant to yellow vein mosaic virus disease are available (Shetty *et al.*, 2013). Four *A. gossypii* resistant okra accessions (VI033805, VI036213, VI051114 and VI033824) and two leafhopper resistant okra accessions (VI041230 and VI033809) have been recently identified by AVRDC. Thus availability and cultivation of similar locally available resistant accessions could substantially reduce pest incidences on okra. Other control methods mentioned in previous sections for aphid, whitefly and leafhopper can be adapted for okra.

8.7.2. Sex pheromone traps can be used to monitor the population of *Earias* spp. (Hall *et al.*, 1980; Cork *et al.*, 1985; 1988; Hamed and Nadeem, 2010).

8.7.3. Biopesticides including *B. thuringiensis* and neem, and novel pesticides such as spinosad are highly effective in reducing the damage caused by *Earias* spp. (Mirmoayedi *et al.*, 2010; Tripathi and Maurya, 2011).

8.7.4. Insecticide applications may be necessary if large flea beetle populations are present at the early stage of crop growth. Foliar insecticides are recommended for quick control of large populations attacking vulnerable seedlings (Varela and Seif, 2004). Since development of insecticide resistance has been reported in *Earias* spp. (Satpute *et al.*, 2003), proper pesticide rotation and windows should be followed to minimize the risk.

8.8. Alliums, *Allium* spp. (Asparagales: Amaryllidaceae)

Onions and garlic are the two most important alliums in the developing world. Onion thrips, *T. tabaci,* is the key pests in most onion production regions of the world. Immature and adult thrips feed on onion foliage by rasping and sucking the juice. The removal of chlorophyll leads to the development of white to silvery patches or streaks, and eventually the damaged leaf area dries up. Thrips feeding can reduce bulb yields by 30-50% and losses can be substantially high if thrips transmit *Iris yellow spot virus*, or create damage that permits other pathogens to infect the crop (Nault *et al.*, 2012). Beet armyworm (*Spodoptera exigua* Hubner) and common armyworm (*S. litura*) are serious defoliators of onions and shallot in Asia. The larvae either feed on the leaf surface or in most cases bore holes in the leaf cuticle and feed from inside the tubular leaves.

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8.8.1. *Allium cepa* L. and *A. fistulosum* L. accessions (AC 570, AF 204, AF 218 and TA 246) are resistant to *S. exigua* (AVRDC, 1997; 1998). Five other onion accessions (AC 521, AC 525, AC 584, TA 189 and TA 243) are resistant to thrips (AVRDC, 1998). Cultivation of similar locally available resistant accessions could substantially reduce pest incidence on onions.

8.8.2. Growing castor as a trap crop on the borders of onion plots reduces the infestation of *S. litura* on onions. Castor should be sown one month before transplanting onions (AVRDC, 1996a). Intercropping of onion with subterranean clover substantially reduces thrips damage. Onions should be planted 30 days after sowing the subterranean clover (AVRDC, 1997).

8.8.3. Onion thrips prefer older plants and infestation is critical during the bulb enlargement stage. Keeping soil moist during this stage helps to reduce onion thrips damage. More irrigation combined with straw mulching reduces thrips damage in onions (AVRDC, 1999).

8.8.4. Sex pheromone traps can be used to monitor and mass-trap *S. litura* and *S. exigua* adults. Installing pheromone traps baited with only one [(Z, E)-9, 12-tetradecadienyl acetate] of the four components of

S. exigua sex pheromone results in mating disruption of both *S. exigua* and *S. litura* (AVRDC, 1997). Traps baited with kairomonal attractants (*e.g.,* p-anisaldehyde and salicylaldehyde) show considerable influence in attracting onion thrips, and could be used as a monitoring and/or mass-trapping tool (AVRDC, 1996a).

8.8.5. Biopesticides and bio-control options noted for *S. litura* above can be adapted for both *S. exigua* and *S. litura* on onions.

8.8.6. Since onion thrips can rapidly evolve resistance to insecticides, chemical pesticides should be applied only when needed (Nault *et al*., 2012). For instance, severe thrips infestation during the bulb enlargement period (the period of 10 weeks after transplanting) can lead to substantial yield losses. It is advisable to initiate spraying two weeks before the initiation of bulb enlargement (AVRDC, 2002). This period may vary slightly depending upon latitude, temperature and onion variety used.

8.9 Conclusion

Cultural practices such as crop rotation, clean cultivation, intercropping, trap cropping, mulching and irrigation are effective in reducing the population build-up of major insect and mite pests in tropical vegetable production systems. Healthy seedling production practices should be adopted to postpone the incidence of early season sucking pests, leaf and stem feeders, and delay the transmission of virus diseases by insect vectors. If insect-resistant varieties with desirable horticultural traits are available, farmers should adopt them to avert severe pest damage and substantial yield loss. Colored sticky traps, traps baited with kairomonal attractants or sex pheromone lures should be installed in fields to monitor and/or mass-trap insect pests. Application of chemical pesticides, especially broad-spectrum insecticides, should be avoided early in the growing season to encourage the proliferation of natural enemies, and to avoid the risk of resurgence of secondary pests. Inoculative releases of parasitoids should be made in newer regions where they are not present. Inundative releases of selected egg parasitoids also provide sufficient control of lepdiopteran pests on vegetables. The use of biopesticides including *B. thuringiensis*, *M. anisopliae*, *B. bassiana*, NPVs and neem should be increased. Chemical pesticides should be properly integrated with other components of pest management in vegetable crops; for example, it is important to choose selective pesticides, and follow a mode of action rotation with a window approach to effectively curtail development of resistance in the target insects. Novel pesticide deployment strategies such as seed treatment, seedling dips, soil drenching or spot application will reduce the direct contact of toxic chemical pesticides on biocontrol agents and pollinators.

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