Q1 IPM for tropical crops: soyabeans

E.A. Heinrichs[1]* and Rangaswamy Muniappan[2]

Address: [1] IPM Innovation Lab, 6517 S. 19th St., Lincoln, NE, USA. [2] IPM Innovation Lab, CIRED, Virginia Tech, 526 Prices Fork Road, Blacksburg, VA, USA.

***Correspondence**: E.A. Heinrichs. Email: eheinrichs2@unl.edu

Received: 29 January 2018
Accepted: 16 October 2018

doi: 10.1079/PAVSNNR201813055

The electronic version of this article is the definitive one. It is located here: http://www.cabi.org/cabreviews

Abstract

Soyabean, because of its importance in food security and wide diversity of uses in industrial applications, is one of the world's most important crops. There are a number of abiotic and biotic constraints that that threaten soyabean production. Soyabean pests are major biotic constraints limiting soyabean production and quality. Crop losses to animal pests, diseases and weeds in soyabeans average 26–29% globally. This review discusses biology, global distribution and plant damage and yield losses in soyabean caused by insect pests, plant diseases, nematodes and weeds. The interactions among insects, weeds and diseases are detailed. A soyabean integrated pest
Q2 management (IPM) package of practices, covering the crop from pre-sowing to harvest, is outlined. The effect of climate changes on arthropod pests, plant diseases and weeds are discussed. The history and evolution of the highly successful soyabean IPM programme in Brazil and the factors that led to its demise are explained.

Q3 Keywords:

Review Methodology: The following databases were searched for relevant articles: Agricola and CAB Abstracts from 1960 on, Plantwise, CABI Invasives, Google Scholar, Google Search, Yahoo Search, EPPO Global Database, Encyclopedia of Life and BugGuide. This was supplemented through personal contacts.

Q4 Introduction

Soyabean, one of the oldest food plants, was domesticated in northeastern China by 1100 BC. Over the next several hundred years the domesticated soyabean (*Glycine max*) spread throughout much of eastern Asia. It grew upright and yielded larger, more digestible seeds. A variety of foods was developed from the soyabean, ranging from soyabean sprouts to steamed raw beans to roasted seeds to soy milk to soy sauce to fermented soyabean paste and cake to soy flour to the commonly eaten curd called tofu (or doufu) [1].

Soyabeans reached the western world by the early 1700s and were first grown in North America by 1804. Benjamin Franklin appears to have been involved in introducing soyabeans from France to Philadelphia at that time. A number of varieties were grown and evaluated in the United States.

Today, the soyabean crop is one of the most important crops worldwide [2]. American soyabean production has been used by the food processing industry – in such foods as margarine, shortening, ice cream, salad dressings and mayonnaise. Industry uses smaller amounts in products including paint, ink, putty, caulking, wallpaper, rubber substitutes, adhesives, fire extinguisher foam, electrical insulation and gasoline (ethanol). The versatile soyabean is a part of everyone's life in developed countries [1].

The crop is grown on about 6% of the world's arable land and since the 1970s; the area in soyabean production has the highest increase compared with any other crop [2]. Global soyabean production was 17 million metric tonnes (MMT) in 1960 and increased to 313 MMT in 2015. Future production is expected to increase more than other crops, due to expanded production area and higher yields.

The USA, Brazil and Argentina dominate global soyabean production [2]. These three countries produced 83% (276 MMT) of world production in 2015. Most of the soyabeans (116 MMT) were produced in the United States, with 99 MMT in Brazil and 61 MMT in Argentina.

China and India produce most of the soyabeans grown in Asia (19 MMT), while only a few are produced in Europe and Africa [3]. In the USA, soyabean is third in production (maize and wheat are first and second).

Nutritional Value of Soyabean

Soyabean plays an important role in food security because it is a highly nutritious crop. In 100 g (raw green soyabeans), soyabeans supply 446 calories and have 9% water, 30% carbohydrates, 20% total fat and 36% protein. Soyabeans are an exceptional source of essential nutrients, providing in a 100-g serving (raw) high contents of the daily value (DV) especially for protein (36% DV), dietary fibre (37%), iron (121%), manganese (120%), phosphorus (101%) and several B vitamins including folate (94%). High contents exist for vitamin K, magnesium, zinc and potassium [4].

Because the soyabean crop is highly nutritious and versatile it offers resources to address world food issues through current and future utilization practices. Future production is expected to increase due to increased demand and with the application of newer genomic technologies the crop has enormous potential to improve dietary quality for people throughout the world whether consumed as a vegetable crop or processed into various soyabean food products [2].

Soyabean Types and Growth Phases

To effectively manage soyabean pests, in an integrated pest management (IPM) context, it is important to be familiar with the soyabean plant and its growth throughout the crop season. Many decisions that a soyabean grower will make depend on the type of soyabean variety and the growth stage of the plant [5, 6].

Soyabean varieties are grouped into 13 maturity groups, depending on the climate and latitude for which they are adapted. These maturity groups are given numbers, with numbers 000, 00, 0 and 1 being adapted to Canada and the northern United States, and numbers VII, VIII and IX being grown in the southern USA. Group X is tropical. Associated with maturity group is the manner in which the stem grows and flowering is initiated. Most northern America adapted cultivars have an indeterminate growth pattern; the terminal bud continues its vegetative activity throughout the growing season and the plant continues to add foliage after flowering. Most cultivars in the southern USA and the tropics have a determinate growth pattern; vegetative growth ceases after flowering. A primary difference between the two, which influences IPM decisions, is that the indeterminate cultivars have the ability to compensate for leaf loss.

For the purpose of managing soyabean pests, growth stage is the most important criterion because the relationship between insect injury and crop damage is dependent on the stage when the injury occurs. Research studies indicate that injury during the vegetative stages is usually not as detrimental to the plant as that during the reproductive stages. The following system is used with ‘V’ representing vegetative stages and ‘R’ reproductive stages:

Vegetative stages	Reproductive stages
VE: emergence	R1: beginning bloom
VC: cotyledon + unfolding unifoliate	R2: full bloom
V1: 1st node trifoliate	R3: beginning pod
V2: 2nd node trifoliate	R4: full pod
V3: 3rd node trifoliate	R5: beginning seed
V4: 4th node trifoliate	R6: full seed
V5: 5th node trifoliate	R7: beginning maturity
V*n*: *N*th node trifoliate	R8: full maturity

Because soyabean response to insects is dependent on growth stage, economic thresholds vary with the stage. Therefore, it is important that growers recognize these developmental stages.

Cropping Systems

Most soyabeans are grown in a crop rotation sequence, typically with a non-legume such as maize, small grains, sorghum or cotton. The yield of the non-legume is improved because of the left-over nitrogen from the soyabean root nodules. In addition, disease, pest and weed problems are reduced in rotations compared with growing one crop continuously [1].

Soyabeans are also often grown in a double-cropping system, with two crops being grown in the same year. Winter wheat followed by soyabeans is the most common; snap beans or peas followed by soyabeans is another.

Constraints to Soyabean Production

There are a number of abiotic and biotic constraints that that threaten soyabean production by directly reducing seed yields and/or seed quality. Abiotic constraints include extremes in nutrients, temperatures and moisture. These reduce production directly, but also indirectly through increase in pathogens and pests [2]. Biotic constraints consist primarily of insects, pathogens, nematodes and weeds.

Pathogens and pests of soyabeans infect and/or attack all parts of the soyabean plant from roots to seed pods [7]. The extent of economic damage depends on the type of pest/pathogen, plant tissue being attacked, number of plants affected, severity of attack, environmental conditions, host plant susceptibility, plant stress level and stage of plant development [8].

Biotic constraints can result in significant negative impacts on yield. Crop losses to animal pests, diseases and weeds in soyabeans average 26–29% globally [9, 10]. Weeds are the predominant pest group in soyabeans. Weed competition causes 37% loss of attainable production while fungal pathogens, bacterial pathogens, viruses and animal pests (insects, mites, nematodes, rodents, birds and mammals) cause 11, 1, 11, and 11% losses of attainable production, respectively. In India, losses in soyabean to the insect complex were estimated to be 18 and 24% in monocrop and when intercropped with maize, respectively [11]. Soyabean disease loss estimates for the top ten soyabean producing countries (97.6% of the world's soyabean production) indicated that losses were highest Q5 due to the soybean cyst nematode (SCN) (*Heterodera glycines*), brown spot (*Septoria glycines*), charcoal rot (*Macrophomina phaseolina*) and sclerotinia stem rot (*Sclerotinia sclerotiorum*) [12].

Soyabean Insect and Disease Classification

Soyabean insects and diseases can be classified by: (1) crop season (early-, mid- and late-season), (2) plant parts (organs) attacked [root, stem, foliage and fruiting structures (pods)], (3) plant growth stage when attack occurs (VE – emergence to R8 – full maturity), (4) insect and disease taxonomy (insect and disease species) and (5) causal agents for the insect and disease damage (bacteria, fungi, oomycetes, viruses and nematodes).

Soyabean insects

Soyabeans can be attacked by pests, at any stage from the seedling stage to close to harvest, but are most attractive to insects from the flowering stage onwards [13]. Insects damage soyabeans directly by feeding on the various soyabean plant parts and indirectly by transmitting a disease while feeding or predisposing the plant to attack of diseases due to the feeding damage.

Based on their importance the insect pests in Brazil are classified as: (1) key pests, (2) secondary pests (some importance in restricted areas or occurring too early or too late in the crop cycle to cause economic damage), (3) regionally important pests and (4) sporadic pests [14].

Globally, there are hundreds of insect species that feed on the soyabean plant but the majority of them do very little damage. According to Hoffman-Campo *et al.* [15], in Brazil only about 35 insect species are considered as soyabean pests. All of the plant parts are vulnerable to insect attack: leaf buds, cotyledons, leaves, stems, nodules, petioles, roots, pods and developing grain.

Insect damage to soyabeans in field is manifested various types of symptoms. These include (1) gaps in the plant stand due to the pruning of roots, wilting plants, tunnelling into the cotyledon, embryo or hypocotyl due to the feeding of coleopteran grubs (larvae) and the seedcorn maggot; (2) holes in leaves or skeletonized leaves due to the feeding of defoliating insects primarily Lepidoptera and Coleoptera; (3) leaves are distorted and discoloured due to the feeding of aphids and spider mites and (4) pods are damaged by the feeding of the bean leaf beetle, pod borers and stink bugs [16].

Traditionally insects associated with crops, such as soyabean, are grouped according to their feeding behaviour [17]. This classification allows damage estimates, appropriate sampling methods and even preliminary identification [4].

Based on their feeding behaviour they are placed in four groups:

(1) root and below ground feeders,
(2) stem feeders,
(3) foliage feeders and
(4) pod feeders.

Root and below ground feeders (Table 1)
Insects in three orders, Coleoptera, Diptera and Hemiptera are considered to be root and below ground feeders. They feed on soyabean roots and seed.

Grape colaspis is a pest in North America. Adults feed on soyabean trifoliate leaves and larvae prune the roots and root hairs of young seedlings. The grubs move up in the soil to near the surface in late May, remove fine roots and gouge 'channels in the roots of soyabean plants'. The grubs are small white larvae with a brown head capsule and neck shield. Damaged seedlings have few lateral roots and the underground stem may show feeding on the bark. Seedlings may die or be stunted by the feeding of the insect [18].

White grubs: White grubs are polyphagous pests and attack soyabeans in North America and Brazil (Table 1). The larvae are white and 'C'-shaped. The true white grub *Phyllophaga* can be identified by having two, parallel rows of stiff bristles on the underside of its tail end. Annual white grubs *Cyclocephala lurida*, lack this pattern of bristles [19]. Injury to soyabean from white grubs is uncommon. However, second and third-instar true white grub larvae can cause damage to soyabean by feeding on roots and causing stand loss.

True white grubs typically have life cycles that span 3–4 years (known as *semivoltine*) while the annual white grub is *univoltine*. These grubs are not common in soyabean, but in infested fields, larvae can be found feeding on roots throughout the growing season. The adults fly to and feed on foliage of deciduous trees and, during spring, will drop to the ground to deposit eggs. Larvae will feed in the root zone just a few inches from the soil line.

Wireworms: Although economic damage to wireworms in North America is rare, when they are a problem, they can be very destructive and difficult to control. They are usually found attacking soyabeans planted in fields that have been in sod for several years, or the second year following sod [20]. Wireworms can feed on

Table 1. Pests of soyabean and their geographical distribution: insects – root and below ground feeders

Insects	Asia	Africa	Europe	N. America	S. America	Australia
Root and below ground feeders						
Grape colaspis						
Colaspis brunnea				X		
(Coleoptera: Chrysomelidae)						
White grubs						
Phyllophaga spp.					X	
(Coleoptera: Scarabaeidae)						
Southern masked chafer						
Cyclocephala lurida				X		
(Coleoptera: Scarabaeidae)						
Wireworms						
Melanotus spp.				X		
Agriotes mancus				X		
Limonius dubitans				X		
(Coleoptera: Elateridae)						
Seedcorn maggot						
Delia platura				X		
(Diptera: Anthomyiidae)						
Brown root stink bug						
S. castanea					X	
(Hemiptera: Cydnidae)						
Bean leaf beetle						
C. trifurcata				X		
(Coleoptera: Chrysomelidae)						

and damage one or more portions of a soyabean seed, or can completely hollow it out leaving only the seed coat. Wireworms may also cut off small roots or tunnel into the underground portions of young soyabean plants. These plants will appear stunted or wilted [21]. Since the life cycle of some wireworm species may last from 4 to 6 years, larvae may damage successive crops [22].

Seedcorn maggot: Seedcorn maggots may destroy soyabean seeds before they germinate or kill young plants before they emerge from the soil. Maggots will also feed on developing cotyledons, which can destroy the growing tips. Damage is most likely to occur if germination is delayed due to cool wet weather [23]. Disturbed soil with decaying organic matter, such as manure or green plant material (cover crop or weeds), is highly attractive to female flies for oviposition. Seedcorn maggot feeding can result in direct loss by destroying the seed and causing stand loss [22].

Brown root stink bug is a pest of soyabean in South America. It has been intensively studied in Brazil [24]. Both nymphs and adults suck the sap of plants from the roots and cause withering and even plant death. They often occur in fields previously in pastures. In Mato Grosso, Brazil, 120 000 ha of soyabean was infested with brown root stink bug in 1999 resulting in yield losses ranging from 20 to 80%. It is most common in sandy soils but also occurs in clay soils. Damage symptoms include a reduction in plant height and reduced stand due to dead plants, and a reduction in the number of pods per plant. Yield losses were greatest when attack occurred in the early stages of plant development.

Bean leaf beetle – a vector of bean pod mottle virus (BPMV) – is a soyabean pest in North America. The larvae live underground, feeding on soybean roots, particularly nodules. Bean leaf beetle adults feed on the above ground part of soyabean such as cotyledon, leaves and pods (https://wiki.bugwood.org/NPIPM:Cerotoma_trifurcata).

Stem feeders (Table 2)

Insects in four orders attack soyabean stems. The stem feeders bore within the stem leaving feeding tunnels (Coleoptera, Diptera and Lepidoptera) or pierce the stems and suck phloem sap from within the stem (Hemiptera).

Soybean stem borer is a pest in Asia. The small, bluish grey beetle lays its eggs in the petioles of soyabean leaves. The larvae tunnel down to the base of the plants, eventually girdling the stem just above the soil line. Lodging occurs as the plants mature, making harvest difficult and reducing yield.

Soybean stem flies: *Melanogromyza sojae* and *Ophiomyia phaseoli* are widely distributed globally. *M. sojae* is an important soyabean pest in Africa and Asia but does not occur in North America and *O. phaseoli* occurs in all regions except in South America. *M. sojae* flies lay eggs on undersides of leaves. After hatching from the egg, yellowish maggots mine through the leaf tissue towards the mid vein. From here, they work their way down the leaf petiole into the stem where it feeds on the pith. If the infected stem is opened by splitting, a distinct zigzag reddish tunnel can be seen with maggots or pupae inside it. The maggots feed on cortical layers of the stem, and feeding damage may extend

Table 2. Pests of soyabean and their geographical distribution: insects – stem feeders

Insects	Asia	Africa	Europe	N. America	S. America	Australia
Stem feeders						
Soybean girdle beetle *Obereopsis brevis* (Coleoptera: Cerambycidae)	X					
Soybean stem borer *Dectes texanus texanus* (Coleoptera: Cerambycidae)				X		
Lucerne crown borer *Zygrita diva* (Coleoptera: Cerambycidae)						X
Cascudinho da soja *Myochrous armatus* (Coleoptera: Chrysomelidae)					X	
Brazilian soybean stalk weevil *Sternechus subsignatus* (Coleoptera: Curculionidae)					X	
Soybean stem fly *M. sojae* (Diptera: Agromyzidae)	X	X	X		X	X
Soybean stem fly *Ophiomyia phaseoli* (Diptera: Agromyzidae)	X	X	X	X		X
Three-cornered alfalfa hopper *Spissistilus festinus* (Hemiptera: Membracidae)				X		
Kudzu bug *Megacopta cribraria* (Hemiptera: Plataspidae)	X			X		X
Common plataspid stinkbug *M. punctatissima* (Hemiptera: Plataspidae)	X					
Lesser cornstalk borer *Elasmopalpus lignosellus* (Lepidoptera: Pyralidae)				X	X	
Broca das axilas *Crocidosema aporema* (Lepidoptera: Tortricidae)					X	

to the taproot. Before pupation, which takes place inside the stem, the larva makes an exit hole for the emergence of the adult, through the stem's xylem and phloem tissue. It is this damage to the soyabean plant's vascular tissue that affects the plant's growth and reduces yield [25].

Three-cornered alfalfa hopper, a North American pest, is a membracid that prefers leguminous plants such as alfalfa, soyabean, bean, cowpea and sweet clover (https://soybeans.ces.ncsu.edu/three-cornered-alfalfa-hopper/). Adults and late instar nymphs feed on the plant sap by piercing the stem with their needle-like mouthparts and extracting plant juices a few inches above the soil surface. Feeding results in the girdling of the main stem. Typically, an insect will repeatedly feed in a ring-like fashion at the same location on the stem. As the plant grows, a swollen callus generally develops on the stem at the feeding site. Stems may break at the site of the girdle. On large plants, lateral branches, leaf petioles and pod stems are often girdled in a similar fashion. Stem injury on small plants may ultimately cause plants to break and lodge [26].

Kudzu bug: The kudzu bug is an invasive pest that was introduced into North America from Asia (India and China) where it is a native. In the southeastern United States, it was first observed in northeastern Georgia in 2009. It has since spread to several neighbouring states. It is the only representative of the family Plataspidae in North America.

The insect taps through the veins of plants to reach the phloem, using piercing–sucking mouthparts. As a result, injury to plants likely results from nutrient and moisture loss, rather than a direct loss of biomass from removal of plant tissue. On soyabeans, the kudzu bug adults and nymphs feed on stems. Excessive kudzu bug feeding negatively impacts soyabean yield by reducing pods per plant, reducing beans per pod, and/or reducing seed size. In 2011, yield losses of up to 47% were recorded in Georgia on untreated beans on a research station near Midville; only two kudzu bugs were found at this location the previous autumn [27]. Q6

The **Lesser Cornstalk Borer** (LCB) is a lepidopteran species of snout moth. It is found from the southern

United States to Mexico, Central America and South America (Colombia, Venezuela, Brazil, Peru, Argentina and Chile) [28].

The LCB larva is bluish-green with brown or purple bands. When disturbed the larva thrashes wildly. Larvae build a distinctive sand-covered silken tube that is attached to the host plant. LCB damage to soyabean is first noticed as wilting, dead or lodged plants. When seedlings are uprooted, damage at the soil line may consist of an entry hole, girdling or multiple feeding sites. Splitting the stems sometimes reveals that the larva tunnelled into the plant. If plants are older when attacked, LCB feeding may not kill them and damage shows up later when stems break at the soil line from previous injury. This injury can be confused with that of three-cornered alfalfa hopper but LCB injury always occurs at the soil line whereas three-cornered hopper injury occurs higher up on the main stem or lateral branches [29].

The life cycle can take only 3 weeks and multiple generations occur, but soyabean becomes less vulnerable to attack as the plant grows and the stem thickens. LCB thrives on hot, dry sandy soils because the larval cuticle or 'skin' is able to retain body water much better than most insects. As a result, the LCB can thrive when conditions are too extreme for the beneficial insects that normally control it.

Foliage feeders (Table 3)

The 'foliage feeders' refers to a broad category which consists of a number of insect orders including Coleoptera, Diptera, Hemiptera, Lepidoptera, Orthoptera and Thysanoptera, and a mite species (Acarina: Tetranychidae), that feed on soyabean foliage. The group has mouthparts that determine the type of feeding damage caused. The lepidopterous larvae, coleopterous larvae and adults and orthoptera nymphs and adults, have chewing mouthparts which are used to defoliate plants. The Diptera larvae have cephalopharyngeal skeleton mouthparts, the Hemiptera nymphs and adults and mites have piercing–sucking mouthparts that penetrate the foliage and Thysanoptera have rasping–sucking mouthparts.

Soyabeans are able to compensate for large amounts of foliage loss due to insect feeding and often little yield loss is observed [30]. Soyabeans can tolerate up to 33% leaf loss (providing terminal and auxiliary buds are not attacked) without yield loss. Soyabean plants not only continue to put out new leaves at the top to compensate for the feeding but also leaves positioned below the feeding injury sites actually grow larger, increasing surface area, since they are getting more sunlight through the canopy. Soyabeans set a large number of reserve pods and can compensate for insect damage during early podding by diverting energy to fill these reserve pods. However, the most critical stage for soyabeans is beginning bloom (R1) to full pod (R4), when seed development is highly dependent on photo-period [16].

Chrysomelidae: Among the beetle species attacking soyabeans, chrysomelids are the most important in Brazil [31]. Several chrysomelids are soyabean pests in Asia, North and South America and Australia. Of the *Diabrotica* species, the banded cucumber beetle, *Diabrotica balteata* and the spotted cucumber beetle *D. undecimpunctata* (also known as the corn rootworm) are soyabean defoliators in North America and the cucurbit beetle, *D. speciosa* is a soyabean leaf feeder in Brazil. *Diabrotica* are polyphagous feeding on a wide range of hosts in the families Asteraceae, Cucurbitaceae, Fabaceae, Poaceae and Solanaceae. Adults feed on rice foliage while the larvae feed on plant roots.

Leaf miners, *Liriomyza sativae* and *Liriomyza trifolii*: These are a few of the dipterans feeding on soyabean foliage. They are widely distributed except for that *L. sativae* does not occur in Europe. They are polyphagous pests of many vegetable and flower crops. Their preferred hosts tend to be in the Cucurbitaceae, Fabaceae and Solanaceae families [32].

Adults and larvae damage the plant foliage. Feeding punctures appear on the upper side of the leaves as white speckles 0.13–0.15 mm in diameter. Oviposition punctures are smaller (0.05 mm) and are more uniformly round. Foliage punctures caused by female adults during the acts of oviposition or feeding may cause a stippled appearance on foliage, but this damage is slight compared with the leaf mining activity of larvae. The irregular mine increases in width from about 0.25 mm to about 1.5 mm as the larva matures. Larvae are often easily visible within the mine where they remove the mesophyll between the surfaces of the leaf. Their faecal deposits are also evident in the mines [33].

Whiteflies: Among the whitefly species the sweet potato whitefly and the silver-leaf whitefly (SLW) are globally distributed foliage-feeding pests. The SLW B-biotype is a pesticide-resistant strain of the whitefly *Bemisia tabaci*. The SLW is a major global pest of horticultural and agricultural crops throughout the world, except in Antarctica [34] and is highly resistant to most current pesticides.

The SLW, a polyphagous pest of agricultural importance throughout the world, can secrete large amounts of sticky honeydew. Although the whitefly may cause some crop losses, simply by sucking sap when they are very numerous, the major harm they do is indirect. Firstly, like many other sap-sucking Hemiptera, they secrete large amounts of honeydew that support unsightly or harmful infestations of sooty mould. Adult females produce more honeydew than other stages and nymphs produce more honeydew when feeding on stressed plants. Honeydew is not a major problem, but the sooty mould, which develops on honeydew shields, leaves from sunlight and reduces photosynthesis. Secondly, they inject saliva that may harm the plant more than either the mechanical damage of feeding or the growth of the fungi. However, by far their major importance as crop pests is their ability to transmit plant diseases [13].

Table 3. Pests of soyabean and their geographical distribution: insects and mites – foliage feeders

Insects and mites	Asia	Africa	Europe	N. America	S. America	Australia
Foliage feeders						
Western striped cucumber beetle – *Acalymma vittatum* (Coleoptera: Chrysomelidae)				X		
Banded cucumber beetle *D. balteata* (Coleoptera: Chrysomelidae)				X		
Cucurbit beetle *D. speciosa* (Coleoptera: Chrysomelidae)					X	
Spotted cucumber beetle *D. undecimpunctata* (Coleoptera: Chrysomelidae)				X		
Cascudinho verde *Maecolaspis calcarifera* (Coleoptera: Chrysomelidae)					X	
Red-shouldered beetle *Monolepta australis* (Coleoptera: Chrysomelidae)						X
Bean leaf beetle *C. trifurcata* (Coleoptera: Chrysomelidae)				X		
Locust leafminer *Odontota dorsalis* (Coleoptera: Chrysomelidae)				X	X	
Mexican bean beetle *E. varivestis* (Coleoptera: Coccinellidae)				X		
Imported longhorn weevil *Calomycterus setarius* (Coleoptera: Curculionidae)	X			X		
Besouro marrom *Aracanthus mourei* (Coleoptera: Curculionidae)					X	
Blister beetle *Epicauta pestifera* (Coleoptera: Meloidae)				X		
Blister beetle *E. lemniscata* (Coleoptera: Meloidae)				X		
Japanese beetle *P. japonica* (Coleoptera: Scarabaeidae)	X		X	X		
American serpentine leafminer *L. trifolii* (Diptera: Agromyzidae)	X	X	X	X	X	
Leaf miner *L. sativae* (Diptera: Agromyzidae)	X	X		X	X	X
Sweet potato whitefly *B. argentifolii* (*B. tabaci* biotype B) (Hemiptera: Aleyrodidae)	X	X	X	X	X	X
Silverleaf whitefly *B. tabaci* (Hemiptera: Aleyrodidae)	X	X	X	X	X	X
Soybean aphid *A. glycines* (Hemiptera: Aphididae)	X		X	X		X
Green mirid *Creontiades dilutus* (Hemiptera: Miridae)						X
Leaf folder *Hedylepta indicata* (Lepidoptera: Crambidae)	X					
Leaf folder						

Table 3. (Continued)

Insects and mites	Asia	Africa	Europe	N. America	S. America	Australia
Nacoleia vulgalis (Lepidoptera: Crambidae)	X					
Bean leafroller *Omiodes diemenalis* (Lepidoptera: Crambidae)	X	X				X
Saltmarsh caterpillar *Estigmene acrea* (Lepidoptera: Erebidae)				X		
Green cloverworm *Plathypena scabra* (Lepidoptera: Erebidae)				X		
Bihar hairy caterpillar *Spilosoma obliqua* (Lepidoptera: Erebidae)	X					
Leaf miner *Aproaerema modicella* (Lepidoptera: Gelechiidae)	X	X				
Australian soybean moth *A. simplexella* (Lepidoptera: Gelechiidae)	X					X
Silver-spotted skipper *Epargyreus clarus* (Lepidoptera: Hesperiidae)				X		
Velvetbean caterpillar *A. gemmatalis* (Lepidoptera: Noctuidae)				X	X	
Green semilooper *Chrysodeixis acuta* (Lepidoptera: Noctuidae)	X					
Tobacco looper *C. argentifera* (Lepidoptera: Noctuidae)						X
Green looper caterpillar *C. eriosoma* (Lepidoptera: Noctuidae)	X		X	X		X
Soybean looper *C. includens* (Lepidoptera: Noctuidae)				X	X	X
Soybean looper *Thysanoplusia orichalcea* (Lepidoptera: Noctuidae)	X	X	X			X
Green semilooper *Diachrysia orichalcea* (Lepidoptera: Noctuidae)	X					
Green semilooper *Gesonia gemma* (Lepidoptera: Noctuidae)						
Soybean looper *P. includens* (Lepidoptera: Noctuidae)				X	X	
Southern armyworm *S. eridania* (Lepidoptera: Noctuidae)				X	X	
Beet armyworm *S. exigua* (Lepidoptera: Noctuidae)	X	X	X	X		X
Autumn armyworm *S. frugiperda* (Lepidoptera: Noctuidae)	X	X		X	X	
Yellow-striped armyworm *S. ornithogalli* (Lepidoptera: Noctuidae)				X		
Common cutworm						

Table 3. (Continued)

Insects and mites	Asia	Africa	Europe	N. America	S. America	Australia
S. litura (Lepidoptera: Noctuidae)	X					X
Grasshoppers						
Melanoplus spp. (Orthoptera: Acrididae)				X		
Soybean thrips						
Neohydatothrips variabilis (Thysanoptera: Thripidae)				X		
Two-spotted spider mite						
T. urticae (Acarina: Tetranychidae)	X	X	X	X	X	X

Feeding by the SLW manifests itself in several ways [13]: (1) SLW can reduce plant vigour and yield by the sheer weight of numbers removing large amounts of plant photosynthate from the leaves; (2) severe infestations on young plants can stunt plant growth and greatly reduce a crop's yield potential; (3) later infestations can reduce the number of pods set, seed size and seed size uniformity, thus reducing yield and quality and (4) in heavily infested soyabeans, both pods and seeds are often unusually pale. While seed colour is unlikely to be of concern in grain soyabeans (harvested seeds being naturally pale), pod and seed discoloration are a major marketing problem where pods are picked green (e.g. vegetable soyabeans). As a rule, the impact of SLW is worst in drought stressed crops.

The SLW is also a vector of a virus of the carlavirus group on soyabean. This virus is responsible for the disease known as 'soybean stem necrosis'. Soyabean plants infected with this virus display necrotic stems, which as symptoms progress may kill the plant [34].

The soybean aphid, a native of Asia and invasive pest, was first observed in the USA in 2000 [35]. It also occurs in Europe and Australia but is not known to occur in Africa and South America (Table 2). In North America *Rhamnus cathartica* (buckthorn) is the primary host. Soyabean is the most important secondary or summer host [36].

Aphids have piercing–sucking mouthparts that suck juices and nutrients from the plant. Removal of plant sap by feeding on the leaves causes stunting, leaf distortion and reduced pod set. An additional threat is the ability of the aphid to transmit soyabean viruses [37].

Honeydew excreted by soybean aphids leads to the development of sooty mould that restricts photosynthesis [2]. Yield loss due to soybean aphid feeding is greatest when soyabeans are in the early R stages (R1–R2), the stages when flowers abort and impact pod establishment. Peak infestations during the pod fill stage (R3), and beyond, can result in smaller seed size and a reduction in seed quality.

An economic analysis of the impact on soyabean production predicted that, without effective plant resistance, US$3.6 to $4.9 billion in soyabean production could be lost annually depending on the cost of insecticide applications, the size of the aphid outbreak and the price elasticity of the soyabean supply [38].

Lepidopteran defoliators: There are numerous lepidopteran species that are key or secondary defoliators of soyabean plants. These include leaf folders, leafrollers, leaf miners, loopers, semi-loopers, armyworms, cutworms, various and 'caterpillar' species. A few grasshopper and thrips species are occasional pests feeding on soyabean foliage (Table 3).

Velvetbean caterpillar: It is considered as the 'main' soyabean pest in Brazil [39] and one of the most important defoliators occurring in soyabeans from Argentina to the southeastern United States [40]. It is the most damaging foliage-feeding pest of soyabean in Florida and the southeastern USA [41].

The occurrence of this species is restricted to the American continent, where if feeds on various crops, especially soyabean [42]. The velvetbean caterpillar is a permanent inhabitant of tropical America and migrates northwards into the southeastern United States every year. The caterpillar overwinters in the southern tip of Florida and moves north during the summer months. *Anticarsia gemmatalis* is an annual problem from June through September in Florida, Georgia and Alabama.

Velvetbean caterpillars cause damage by consuming foliage. To complete its larval development, the velvetbean caterpillar consumes from 85 to 150 cm^2 of leaf area but the majority of this consumption is performed by larvae from the 4th to 6th instar, which are caterpillars >1.5 cm [34].

Newly hatched larvae strip the leaf, beginning with the lower epidermis and mesophyll. They continue feeding until the end of the second instar, eating all the soft leaf material and leaving only the veins intact [43]. Eventually the velvetbean caterpillar consumes the entire leaf. Once the upper leaves and lower leaves have been consumed, foliage in the middle and lower canopy is consumed and complete defoliation may result [44]. The velvetbean caterpillar may also attack tender stems, buds and small bean pods.

Soybean thrips differ from the other foliage feeders in the way they feed on leaves. Thrips mouthparts make them unique in that they rasp and puncture plant cells then suck up the exuding sap. Their appearance, biology and feeding damage has been described in [45].

Thrips are minute, slender-bodied insects ranging from 0.8 to 5 mm in length. The immatures are yellow, while adults are black with yellow bands. Their wings, when present, are fringed with long hairs. Although rarely noticed, thrips are probably the most numerous insects in soyabean.

The adult female inserts oblong eggs singly into the leaves of the plant upon which she feeds. Young thrips go through four wingless stages between hatching and adulthood. An entire life cycle requires only 2–4 weeks. Thrips are present throughout the summer in the USA.

Thrips make tiny, linear, pale-coloured scars on soyabean leaves where they penetrate individual leaf cells and feed on the contents. They usually feed on the undersides of leaves, much of which occurs along the veins. When leaves are heavily infested, the feeding scars may be so numerous that a mottling look appears along and between the leaf veins and leaves may become crinkled in appearance. Another indication of their activity is the presence of black 'tar spots' on the leaves. This is thrips excrement and the descriptive name is quite accurate.

Soyabean is particularly susceptible to thrips damage early in the growing season from growth stages VE to V6. Although thrips are a common pest of soyabeans in the United States, they rarely cause economic damage. Nevertheless, yields may be reduced if plants are under moisture stress and thrips populations are high.

Two-spotted spider mites are globally distributed throughout the world (Table 3). During feeding the mites penetrate the plant foliage/leaves with their mouth stylets and suck out the cell contents. The result of this damage to leaf tissue is reduced chlorophyll content and reduced photosynthesis, carbon dioxide assimilation and transpiration. Feeding by the two-spotted spider mite causes pale spots to appear on leaves. As infestations become more severe, leaves appear bronzed or silvery, become brittle, and may fall prematurely. The mites spin webbing, which can cover all the surfaces of the plant. Plants can be rapidly killed this mite (https://www.cabi.org/isc/datasheet/53366).

Pod feeders (Table 4)

Insects in the orders Coleoptera (beetles), Lepidoptera (armyworms) and Hemiptera (stink bugs) damage soyabean pods. The Coleoptera larvae and adults and Lepidoptera larvae have chewing mouthparts with which they devour the pods and green beans within. Both the stink bug nymphs and adults have piercing–sucking mouthparts for removing plant fluids [16].

Among the lepidopterous pod feeders, the lima bean pod borer (*Maruca vitrata*), pod borer (*Helicoverpa armigera*) and the pea pod borer (*Etiella zinckenella*) are widely distributed globally.

Legume pod borer is considered the most serious pest of food legumes in tropical Asia, sub-Saharan Africa, South America, North America, Australia and the Pacific [46]. Its bionomics and management on cowpea have been reported [47]. It is an invasive species, and a pantropical insect pest (Table 4) of leguminous crops e.g. cowpea, pigeonpea, mung bean, common bean, soyabean, adzuki bean, groundnut, hyacinth bean, and field peas, country bean, broad bean, kidney bean and lima bean. The legume pod borer is a serious pest of grain legumes in the tropics and sub-tropics because of its extensive host range, and destructiveness.

The importance of it as a pest on grain legumes results from its early establishment on the crop. The larvae web the leaves and inflorescence, and feed inside flowers, flower buds and pods. This typical feeding habit protects the larvae from natural enemies and other adverse factors, including insecticides. The successful establishment of this pest at the flower bud stage is significant in relation to subsequent damage, reduction in grain yield and efficiency of control. Third to fifth-instar larvae are capable of boring into the pods and consuming the developing grains.

Lima bean pod borer occurs in Asia, sub-Saharan Africa, South America, North America, Australia and Europe. Although it has been reported on several legume crops, it causes significant yield losses in lentil, peas and soyabean [48]. Infestation of soyabean plants usually starts late in the season. The larvae feed on floral buds, flowers and pods by making rough and irregular incisions. The pods may have several entry holes. The larvae feed on the seeds inside the pods. Often the damaged pods and inflorescence show light coloured frass and loosely spun webs. In Indonesia, *E. zinckenella* can cause severe damage to the pods and yield losses can reach 80% [49].

***Helicoverpa* species:** Three *Helicoverpa* species are pod feeders (Table 4). The pod borer *H. armigera* occurs in all regions, recently invading North America (Florida). The native budworm *H. punctigera* is a pest in Australia and the American cotton bollworm *H. zea* attacks soyabean pods in North and South America. All have similar feeding habits.

The **gram pod borer**, *H. armigera*, is a polyphagous and highly mobile insect and it is a pest of economic importance on many agricultural and horticultural crops. It is a major pest on a number of crops, including cotton, tobacco, corn (maize), sorghum, sunflower, soyabean, lucerne and pepper [48]. It has been recorded on 180 cultivated and wild plant species in at least 45 families. Occasionally it causes significant yield losses in mungbean, vegetable soyabean, chickpea and pigeon pea.

The neonate larvae feed on the surfaces of leaves or floral buds. However, the grown-up larvae prefer to feed on the contents of reproductive parts such as floral buds, flowers and young fruits. The larvae make the holes on these reproductive parts and feed inside by thrusting their head inside; hence, the holes are circular and often surrounded by faecal pellets. Later, the larva feeds on most of the inner contents of the pod and damaged pods may become deformed [48].

H. armigera is an important soyabean pest in Australia where it can severely damage all crop stages and all plant

Table 4. Pests of soyabean and their geographical distribution: insects – pod feeders

Insects	Asia	Africa	Europe	N. America	S. America	Australia
Pod feeders						
Bean leaf beetle *C. trifurcata* (Coleoptera: Chrysomelidae)				X		
Legume pod borer *M. vitrata* (Lepidoptera: Crambidae)	X	X	X	X	X	X
Gram pod borer *H. armigera* (Lepidoptera: Noctuidae)	X	X	X	X	X	X
Native budworm *H. punctigera* (Lepidoptera: Noctuidae)						X
American cotton bollworm *H. zea* (Lepidoptera: Noctuidae)				X	X	
Tobacco budworm *Heliothis virescens* (Lepidoptera: Noctuidae)				X	X	
Southern armyworm *S. eridania* (Lepidoptera: Noctuidae)		X		X	X	
Garden armyworm *S. latifascia* (Lepidoptera: Noctuidae)				X	X	
Lima bean pod borer *E. zinckenella* (Lepidoptera: Pyralidae)	X	X	X	X	X	X
Green stink bug *Acrosternum hilare* (Hemiptera: Pentatomidae)				X		
Southern green stink bug *N. viridula* (Hemiptera: Pentatomidae)	X	X	X	X	X	X
Neotropical brown stink bug *E. heros* (Hemiptera: Pentatomidae)					X	
Brown stink bug *E. servus* (Hemiptera: Pentatomidae)				X		
Redbanded shield bug *P. grossi* (Hemiptera: Pentatomidae)	X	X				X
Redbanded stink bug *P. guildinii* (Hemiptera: Pentatomidae)				X	X	
Large brown bean bug *Riptortus serripes* (Hemiptera: Alydidae)						X
Small brown bean bug *Melancanthus scutellaris* (Hemiptera: Alydidae)						X
Brownshield bug *Dictyotus caenosus* (Hemiptera: Pentatomidae)						X
Green-belly stink bug *D. furcatus* (Hemiptera: Pentatomidae)					X	
Green and brown stink bug *Edessa meditabunda* (Hemiptera: Pentatomidae)					X	
Brown marmorated stink bug *Halyomorpha halys*	X		X	X	X	

Q23

Table 4. (Continued)

Insects	Asia	Africa	Europe	N. America	S. America	Australia
(Hemiptera: Pentatomidae)						
Neotropical red-shouldered stink bug *Thyanta perditor* (Hemiptera: Pentatomidae)				X	X	

parts of soyabeans. Of the summer legumes, soyabeans are the most attractive to *Helicoverpa* during the vegetative stage and can even be damaged during the seedling stage. In sub-coastal and inland southern Queensland, summer legumes are at greatest risk from *H. armigera* from mid-December onwards [13].

Stink bugs: While pod-sucking bugs start breeding as soon as they move into flowering crops, nymphs must feed on pods to complete their development [13]. Both adults and nymphs have piercing–sucking mouthparts for removing plant fluids. Stink bugs feed directly on pods and developing seeds. However, their damage is difficult to assess because their mouthparts leave no obvious feeding scars on the surface of the pod. Instead, they inject enzymes into the seeds, causing the seed to shrivel [13]. The feeding wound provides an avenue for diseases to enter into the pod.

Seeds damaged by stink bugs during early pod-fill are often lost at harvest, or are graded out post-harvest, as they are lighter than undamaged seeds. Crops remain susceptible to late bug damage until the pods harden just prior to harvest [16]. Indirect effects can include delayed maturity (green bean syndrome) of injured plants, though stink bugs are not the only cause for green bean syndrome.

There are many stink bug species feeding on soyabeans globally [50]. The list in Table 4 is a representation of the important species in the six regions of the world. The southern green stink bug *Nezara viridula* is widely distributed and an important pest of soyabeans globally. *N. viridula*, the Neotropical brown stink bug *Euschistus heros*, and the redbanded stink bug *Piezodorus guildinii*, are major pests in Brazil and represent 98% of the total stink bug population [51]. The brown marmorated stink bug (BMSB) is an invasive pest occurring in Asia, Europe and North America with potential to spread to other regions.

Southern green stink bug, a native to Ethiopia, is a cosmopolitan and highly polyphagous pest species feeding on more than 52 documented host plants, including ornamentals, weeds and cultivated crops, in 30 plant families [52]. Although it causes economic damage to many crop species, it mainly feeds on legumes. It is an invasive pest and was first reported in northeastern Brazil in 2001 [53]. This species is the most damaging pod-sucking bug in soyabeans due to its abundance, widespread distribution, rate of damage and rate of reproduction. Nymphs are less damaging than adults. While first instar nymphs cause no damage, subsequent instars are progressively more damaging, with the fifth and final instar being nearly as damaging as adults are.

The damage symptoms are similar to other pod bugs, causing drying of shoots, shrivelled pods and seeds. Bug damaged seeds are frequently discoloured, either directly or due to the yeast-spot disease, *Nematospora coryli* [54], which gains entry when pods are pierced by bugs.

It can even damage seeds in 'close-to-harvest' pods (i.e. pods that have hardened prior to harvest). Seeds damaged in older pods are blemished and difficult to grade out, reducing harvested seed quality, particularly that destined for human consumption (edibles). Damage to young pods causes deformed and shrivelled seeds and reduce yield. Bug-damaged seeds have increased protein content but a shorter storage life (due to increased rancidity). Bug damage also reduces seed oil content.

Neotropical Brown Stink Bug is a Neotropical pentatomid occurring in South America. Most reports refer to its distribution in Brazil where it is a major component of the pentatomid-pest complex on soyabean. Initially considered a secondary pest, *E. Heros* has increased in numbers and may inflict severe damage to soyabean. In the southern-most state of Rio Grande do Sul (RS) in Brazil *E. heros* is now the most abundant species of pentatomid on soyabean, reaching over 80% of the total number of stink bugs collected in Passo Fundo, RS (A. R. Panizzi, personal communication).

Beyond soyabean, this insect feeds on several other plants (in 11 families) in Brazil, including species of Leguminosae, Fabaceae, Solanaceae, Brassicaceae and Compositae [55]. Even though it is polyphagous and, therefore, able to colonize alternate hosts during the mild winter of northern Paraná state, *E. heros* was found to overwinter underneath dead leaves in this area. This bug is known to accumulate lipids during overwintering that allow survivorship during non-feeding periods, and to break dormancy with the start of favourable weather conditions. During summer, with the availability of the great number of weeds associated with the soyabean crop, this bug also feeds on some weeds.

Redbanded stink bug has a wide Neotropical distribution (Table 4) that ranges from Argentina to the southern United States [50]. Since the late 1970s, the redbanded stink bug has replaced the southern green stink bug as the principal stink bug pest in portions of Brazil [56]. Redbanded stink bugs appear to be more adapted to warmer climates.

It has existed in Florida (USA) for many years without being a serious pest. However, it has recently become a major pest of soyabean in Alabama, Georgia, Louisiana, Mississippi, South Carolina and Texas [55]. It was first identified as a pest of Louisiana (USA) soyabean in 2000 [57]. In a survey conducted in Louisiana the redbanded stink bug comprised the largest percentage (54%) of the total stink bug complex collected at five survey sites followed by *N. viridula* (27%), and, *Euschistus servus* (7%).

Compared with *N. viridula*, *P. guildinii* feeds on fewer plant species, being confined mostly to legumes (Fabaceae) [58]. Plants of the genus *Indigofera* seem to be preferred [59]. However, other species of cultivated and non-cultivated plants of different families are at various times used by this pentatomid, either as a source of nutrients or water, or as shelter. The list of plants on which *P. guildinii* has been recorded in the neotropics includes 49 plant species belonging to 22 families, of which 24 species were considered reproductive hosts.

Studies in Argentina have shown that the redbanded stink bug has the greatest capacity to damage soyabean among all South American phytophagous stink bugs [60]. *P. guildinii* feeding causes reductions in yield and seed quality and foliar retention. Galileo and Heinrichs [61, 62] evaluated redbanded stink bug effects on soyabean pod production seed quality in Brazil. The bugs were infested at zero, two, four, six, and ten insects/1.5 row ft. during the growth stages R2–R4, R5, R6–R7 and R2–R7. No significant reduction in yields was detected at two-ten stink bugs/1.5 row ft. during R2–R4 growth stages. During the R5 growth stage, only two stink bugs/1.5 row ft. significantly reduced yields. Infestations during R6–R7 growth stages required six stink bugs/1.5 row ft. to significantly reduce yields [60].

Significant reductions in seed production only occurred when ≥four redbanded stink bugs/1.5 row ft. were infested during R2–R7 growth stages. All treatments had significantly more damaged seed compared with the non-infested control during all growth stages except for R2–R4. Percent of seed damaged ranged from 39 (two insects/1.5 row ft.) to 72% (10 insects/1.5 row ft.) [62].

In Brazil, delayed soyabean maturity (foliar retention) has been associated with redbanded stink bug infestations in numerous reports. Panizzi et al. [63] reported that soyabean retained green leaves when redbanded stink bugs infested soyabean during pod development and pod fill stages (R3–R6). Heinrichs [64] and Galileo and Heinrichs [65] reported excessive green foliage retention when stink bug populations were ≥eight insects/3 ft. row during early seed development, but similar populations at R6 did not delay maturity.

Brown marmorated stink bug also known simply as the 'stink bug', is a native to China, Japan, Korea and Taiwan [66]. Currently it is widespread in Europe, and recently has been found in South America [67] (Table 4). In Japan it is a pest to soyabean and fruit crops. In the USA, the BMSB feeds on a wide range of fruits, vegetables and other host plants including peaches, apples, green beans, soyabeans, sweet corn, cherries, raspberries and pears [68]. An invasive pest, the BMSB was accidentally introduced in the United States from China or Japan. It is believed to have hitched a ride as a stowaway in packing crates or on various types of machinery.

The (BMSB), which was first identified in 2001 in Allentown, PA, USA, was found infesting soyabean fields in parts of Virginia in 2011 [69]. The BMSB, similar to the native stink bug species, feeds directly on developing soyabean pods and seeds. If the damage occurs very early in seed development, pods will be flat and brown, but will still be attached to the plant and easy to see. If damage occurs later in seed development, pods will appear yellow and speckled, and opening the pod will reveal damaged, crinkled, stained seed. One visual cue of stink bug damage to soyabean crops is the 'stay green' effect, where damaged soyabean plants stay green late into season, while other plants in the field die off normally. One can usually tell that a soyabean field is infected because stink bugs are known for their 'edge effect', in which they tend to infest crops 30–40 ft. from the edge of the field.

Heavy infestations also seem to be associated with fields with wooded borders, especially if there are concentrations of the invasive weed, Tree-of-Heaven (*Ailanthus altissima*). Both BMSB and Tree-of Heaven are native to China and other parts of Asia. The BMSB seems to be strongly attracted to that host, especially when the trees are putting out their seed clusters. Not coincidently, the north-central piedmont area of Virginia, where the highest densities of BMSB are found in soyabean, is the area with the highest concentration of Tree-of-Heaven.

Soyabean diseases

Soyabean diseases are major constraints to production in all soyabean-producing regions of the world. The first *Soybean Disease Compendium* [70] covered 50 diseases whereas the latest edition lists more than 300 diseases [7]. The increase in the number of diseases and their expansion are a result of the intense production and increased acreage in new regions of the world [8].

Diseases tend to be geographically and environmentally restricted [2]. Some diseases such as soyabean rust, *Phakopsora pachyrhizi*, are explosive by producing copious amounts of spores and are widely distributed, whereas red leaf blotch, *Phoma glycinicola*, is restricted to a few countries in Africa (Table 5). Diseases based on the season of the soyabean crop consist of early-, mid- and late season diseases. Important and widely distributed early season diseases are bacterial pustule, *Xanthomonas axonopodis* pv. *glycine*, sclerotinia stem rot, *Rhizoctonia solani*, and seedling blight and root and stem rot, *Phytophthora sojae*; diseases attacking at mid-season are the Asian soyabean rust, *P. pachyrhizi*, and soyabean mosaic virus; and late season diseases are charcoal rot, *M. phaseolina*, anthracnose/pod

Table 5. Pests of soybean and their geographical distribution: diseases bacteria and fungi

Diseases	Asia	Africa	Europe	N. America	S. America	Australia
Bacteria						
Bacterial blight						
P. savastanoi pv. *glycinea*	X	X	X	X	X	X
Bacterial wilt						
Curtobacterium flaccumfaciens pv. *Flaccumfaciens*		X		X	X	X
Bacterial pustule						
X. axonopodis pv. *glycines*	X	X	X	X	X	X
Wildfire						
P. syringae pv. *tabaci*	X	X	X	X	X	X
Fungi						
Brown spot						
S. glycines	X		X	X	X	
Cercospora leaf blight						
C. kikuchii	X	X	X	X	X	X
Charcoal rot						
M. phaseolina	X	X	X	X	X	X
Downy mildew						
Peronospora manshurica	X	X	X	X	X	X
Frogeye leaf spot						
C. sojina	X	X	X	X	X	X
Rhizoctonia stem rot						
R. solani	X	X	X	X	X	X
Asian soybean rust						
P. pachyrhizi	X	X	X	X	X	X
New World soybean rust						
P. meibomiae				X	X	
Sclerotinia stem rot						
S. sclerotiorum	X	X	X	X	X	X
Target leaf spot						
C. cassiicola	X	X		X	X	
Pod and stem blight						
D. phaseolorum var. *sojae*	X	X	X	X	X	X
Stem canker						
Diaporthe var. *meridionalis*		X	X	X	X	
Southern blight						
S. rolfsii	X	X	X	X	X	
Powdery mildew						
Microsphaera diffusa	X		X	X	X	X
Alternaria leaf spot						
Alternaria tenuissima	X	X	X			
Myrothecium leaf spot						
Myrothecium roridum	X					
Anthracnose/pod blight						
C. truncatum	X	X	X	X	X	X
Sudden death syndrome						
F. virguliforme (*a.k.a. F. solani f.* sp. *glycines*)				X		
Sudden death syndrome						
F. tucumaniae					X	
Fusarium seedling/collar rot						
F. equiseti	X					
Red leaf blotch						
P. glycinicola		X				

blight, *Colletotrichum truncatum* and the soyabean cyst nematode, *H. glycines*.

The pathogens of soyabean diseases are bacteria, fungi, oomycetes, viruses and nematodes (Tables 5–7). These pathogens cause visible disease symptoms on the entire plant, or on individual plant parts such as roots, stems, leaves and pods. A diagnostic guide designed to provide soyabean growers, agronomists, pest management specialists and others a means to diagnose soyabean diseases, as they are observed in soyabean fields, has recently been published [71]. The Field Guide to African Soyabean Diseases and Pests, the first of its kind, highlights over

Table 6. Pests of soyabean and their geographical distribution: diseases – Oomycetes and viruses

Diseases	Asia	Africa	Europe	N. America	S. America	Australia
Oomycetes						
Phytophthora rot						
P. megasperma	X		X	X	X	
Phytophthora seedling blight and root and stem rot						
P. sojae	X	X	X	X	X	X
Viruses						
Cowpea mild mottle virus						
Genus *Carlavirus*	X	X				
Bean pod mottle virus						
Genus *Comovirus*	X	X		X	X	
Alfalfa mosaic virus						
Genus *Alfamovirus*	X	X	X	X	X	X
Cucumber mosaic virus						
Genus *Cucumovirus*	X	X	X	X	X	X
Southern bean mosaic virus						
Genus *Sobemovirus*	X	X	X	X	X	
Soybean mosaic virus						
Genus *Potyvirus*	X	X	X	X	X	X
Soybean vein necrosis virus						
Genus *Tospovirus*				X		
Soybean dwarf virus	X	X		X		X
Genus *Luteovirus*						
Tobacco ringspot virus	X	X		X	X	X
Genus *Nepovirus*						
Tobacco streak virus	X	X	X	X	X	X
Genus *Ilarvirus*						
Peanut stripe virus	X	X		X		
Genus *Potyvirus*						
Indonesian soybean dwarf virus	X					
Genus *Luteovirus*						
Mungbean yellow mosaic virus						
Genus *Geminivirus*	X					
Soybean crinkle leaf virus						
Genus *Geminivirus*	X					

20 diseases commonly found in many soyabean production areas as well as those that may be unique to Africa. The online guide is available at: http://soybeaninnovationlab.illinois.edu/soybean-disease-diagnostic-guide.

Bacteria

Three bacterial diseases – blight, pustule and wildfire – attack soyabeans wherever they are grown (Table 5), if the weather conditions are favourable for their development. Severe infection by any one of the diseases may cause premature defoliation. In most years in Illinois, USA, bacterial blight and pustule can be found in 40–90% of the soyabean fields in the state [72]. The wildfire disease is more serious in the southern United States. The prevalence and the severity of these diseases fluctuate from year to year, depending on weather conditions. Yield losses range from zero to 15% with 1–2% common. Like many other diseases of soyabeans, bacterial diseases are more severe in wet years than in dry ones.

Bacterial blight caused by *Pseudomonas savastanoi* pv. *glycinea* is commonly found in cool, temperate soyabean-growing regions worldwide. Losses range from minimal to 18% in Korea and the USA [73]. The disease has not caused significant economic losses in most areas of the world due to resistance (four genes identified) or plant tolerance. In the Philippines, infestation of plants with red spider mites (*Tetranychus telarius*) appeared to impart resistance or immunity of soyabean to *P. savastanoi* pv. *glycinea* [73].

This is the most common bacterial disease of soyabeans in the major USA soyabean producing state of Illinois. Cool, rainy weather in June and July, accompanied by strong winds, favours the development of bacterial blight. It ceases to develop during hot, dry weather. Bacterial blight appears on soyabean leaves in June or early July as small, angular, yellow to light brown spots (lesions) with water-soaked centres. As the tissues die, the centres of the lesions soon turn dark reddish brown to black and usually are surrounded by a water-soaked margin bordered by a yellowish green halo. Infected young leaves frequently are distorted, stunted and yellowish (chlorotic).

Bacterial pustule is caused by the bacterium *X. axonopodis* pv. *glycines*. In North America, this disease is typically present on the upper leaves. Initial infection results in the development of tiny pale green spots on the new leaves. This disease can easily be confused with soyabean rust. Mature soyabean rust pustules have a small

Table 7. Pests of soyabean and their geographical distribution: diseases – nematodes

Diseases	Asia	Africa	Europe	N. America	S. America	Australia
Nematodes						
Peanut pod						
Ditylenchus africanus		X				
Soybean cyst						
H. glycines	X	X	X	X	X	
Columbia lance nematode						
H. columbus	X			X		
Peanut root-knot						
M. arenaria	X	X	X	X	X	X
Root-knot						
M. ethiopica		X	X		X	
Root-knot						
M. hapla	X	X	X	X	X	X
Root-knot						
M. incognita	X	X		X		X
Root-knot						
M. javanica	X	X	X	X	X	X
Root-lesion						
P. brachyurus	X	X	X	X	X	X
Root-lesion						
P. crenatus	X	X	X	X	X	X
Root-lesion						
P. neglectus		X		X		
Lesion nematode						
P. scribneri				X		
Root-lesion						
P. teres		X				
Root-lesion						
P. thornei	X	X	X	X	X	X
Root-lesion						
P. zeae		X		X		X
Reniform						
R. parvus		X				X
Reniform						
Rotylenchulus reniformis	X	X	X	X	X	X
Reniform						
R. unisexus		X				X
British spiral						
S. brachyurus	X	X	X	X	X	X
Spiral						
S. truncatum		X				
Common spiral nematode						
H. dihystera	X	X	X	X	X	X
Spiral						
H. pseudorobustus	X	X	X	X	X	X
Sting						
B. longicaudatus				X		

opening at the top for spore release whereas bacterial pustule lesions lack the opening on top and the spores [72].

Like bacterial blight, *Pseudomonas syringae* pv. *glycinea*, bacterial pustule over winters in crop residue and is carried by wind or water droplets from the ground to the plant. In addition, the disease can be spread during cultivation while the foliage is wet. The bacterium will enter the plant through natural openings and wounds. Premature defoliation occurs due to heavy infections, which in turn produces a reduction in seed size and number.

The symptoms of the pustule disease are somewhat similar to those of bacterial blight. In bacterial pustule, many small lesions may merge – producing large, irregular, mottled brown, dead areas with yellowish margins. The leaves become ragged when parts of the brown, dead areas tear away during windy, rainy weather. Severe infection often causes some defoliation.

Wildfire: The name of the disease, wildfire, caused by *P. syringae* pv. *tabaci*, results from the burnt appearance of heavily infected tobacco plants. Wildfire disease rarely occurs in Illinois, USA but is most common in southern USA [72].

In 2006 and 2007, a new bacterial disease, wildfire, was first observed in field-cultivated soyabeans in Munkyung

Q7 City, Korea. The disease caused severe blighting of soyabean leaves [74]. This disease is now widely distributed (Table 5).

The symptoms of wildfire are light brown spots of variable size and shape, which are surrounded by a broad yellow halo. The halo distinguishes wildfire from other bacterial diseases of soyabean. In damp weather, the lesions enlarge and merge forming large, dead areas in the leaf that become dry, tear away, and produce tattered leaves. A bacterial pustule is almost invariably present in the centre of a wildfire lesion. Evidence exists that the presence of bacterial pustule is required for wildfire infection. Bacterial blight lesions also may serve in the same way [72].

Fungi

Most of the soyabean fungal diseases occur in all six geographical regions (Table 5). A few such as sudden death syndrome, *Fusarium virguliforme* and *F. tucumaniae*, Fusarium seedling/collar rot, *F. equiseti* and red leaf blotch *Phoma glycinicola* only occur in North America, South America, Asia and Africa, respectively. The common names of the fungal pathogens are generally based on the plant part attacked and showing symptoms: root, collar, stem, leaf and pod. Common foliar diseases are frogeye leaf spot, *Cercospora sojina*, cercospora leaf blight, *Cercospora kikuchii*, target leaf spot, *Corynespora cassiicola* and soybean rust, *Phakopsora* spp. Diseases attacking the stem are sclerotinia stem rot, *Sclerotinia sclerotium* and stem canker, *Diaporthe* var. *meridionalis*. Some diseases cause damage to several plant parts such as pod and stem blight, *Diaporthe phaseolorum* var. *sojae*. Sudden death syndrome, *Fusarium* spp., attack the roots but foliar symptoms typically appear at soyabean flowering. Soyabean anthracnose, caused by *C. truncatum*, *produces* brown, irregularly shaped spots on stem, pods and petioles [75]. In this section, we focus on a few economically important fungal pathogens.

Rust: There are two closely related fungi that cause rust on soyabean: *P. pachyrhizi*, sometimes referred to as the Asian or Australasian soybean rust pathogen, but which now also occurs in the western hemisphere (Table 5) and *P. meibomiae*, the so-called New World soybean rust pathogen, which is found only in the western hemisphere (Table 5). Except for a few minor characteristics, the two fungi appear morphologically identical, but *P. pachyrhizi* is much more aggressive on soyabean than *P. meibomiae*. To date, *P. meibomiae* has not been documented to cause significant yield losses in South America and has a restricted distribution in North America [76].

Asian soybean rust, an invasive disease caused by *P. pachyrhizi*, is a major disease limiting soyabean production in many areas of the world [2, 77], and has been a serious disease in Asia for many decades [78]. It appeared in Africa in 1997 and in the Americas in 2001. A hurricane, in 2004, probably brought it into the continental USA [79]. Before it was first found in the continental USA, it was considered such a threat that it was listed as a possible weapon of bioterrorism [78]. Soybean rust cannot overwinter in areas with freezing temperatures, but it can spread by wind rapidly over such large distances, its development can be so explosive, and it can cause such rapid loss of leaves, that it is now one of the most feared diseases in the world's soyabean-growing areas [78].

While soybean rust usually begins in the lower canopy, it quickly progresses up the plant until all of the leaves have some level of disease. Severely diseased plants may become completely defoliated. The loss of effective leaf tissue results in yield reductions from both fewer and smaller seed. Yield losses as high as 80% have been reported [79] but the amount of loss depends on when the disease begins and how rapidly it progresses. Besides leaves, soybean rust can also appear on petioles, stems, and even cotyledons, but most rust lesions occur on leaves [78].

The first symptoms of soybean rust, caused by *P. pachyrhizi*, begin as very small brown or brick red spots on leaves. In the field, these spots usually begin in the lower canopy at or after flowering. Lesions remain small (2–5 mm in diameter), but increase in number as the disease progresses. Pustules, called uredines, form in these lesions, mostly on the lower leaf surface, and they can produce many urediniospores. Soybean rust urediniospores are pale yellow-brown to colourless, with an echinulate (short spines) surface ornamentation. This coloration is different from many other rust pathogens whose spores are often reddish-brown (rust coloured). Germination of *P. pachyrhizi* urediniospores occurs through an equatorial (central) pore, producing a germ tube that ends in an appressorium, which the fungus uses to penetrate the host directly or through a stoma. As more and more lesions form on a leaflet, the affected area begins to yellow, and eventually the leaflet falls from the plant [78].

Red leaf blotch: There are only a few known soyabean diseases that are not present in the major soyabean producing countries. Red leaf blotch, caused by the pathogen *Phoma glycinicola*, formerly known as *Dactuliochaeta glycines* [80], is one of them. Symptoms of red leaf blotch include lesions on foliage, petioles, pods and stem [81]. Heavily diseased plants defoliate and senesce prematurely.

Red leaf blotch is only known to occur in Africa (Table 5) and was first reported from there in 1957 [82]. The disease is currently a threat to production in central and southern African countries where it is endemic. *P. glycinicola* is a soil-borne fungus. It is known to infect soyabean and *Neonotonia wightii*, a perennial legume that inhabits the woodlands and grasslands of southern Africa. The fungus is known to occur in Cameroon, Ethiopia, Malawi, Nigeria, Republic of Congo, Rwanda, Uganda, Zambia and Zimbabwe. Most of the published literature about red leaf blotch is from studies completed in Zambia and Zimbabwe, where yield losses of up to 50% were reported [83].

Because the fungus does not produce copious spores like *P. pachyrhizi*, is not seed-borne, and trade of soyabeans out of Africa is limited, the pathogen has not spread to new locations. However, it is a serious, potentially invasive pest.

It is predicted that the pathogen would most likely survive the non-crop season in any of the yet unaffected soyabean production regions worldwide [2]. If the fungus/disease is found in the USA, it may become economically important, because of its potential to cause significant yield losses, the lack of resistant soyabean varieties, and the importance of the soyabean crop to the agro-industry in the USA [84].

Oomycetes

Oomycetes are fungal-like microbes that cause highly destructive plant diseases that affect agriculture, horticulture and forestry. Oomycetes occupy both saprophytic and pathogenic lifestyles, and include some of the most notorious pathogens of plants. Soyabean is a high-value crop that is heavily impacted by oomycete diseases. There are many oomycete species attacking soyabean with the more important species belonging to the *Pythium* and *Phytophthora* genera. Among the oomycetes, the most aggressive plant pathogenic species are found in the *Phytophthora* genera. Phytophthora rot, *Phytophthora megasperma* and phytophthora root and stem rot, *P. sojae* are important and widely distributed diseases (Table 6).

***Phytophthora sojae*:** This disease, prevalent in many soyabean production regions of the world, is an Oomycete that causes a serious root and stem rot of soyabeans. The pathogen can survive for long periods in infected plant tissue or soil as sexual spores called oospores [85]. It can kill and damage seedlings and plants throughout the growing season from the time of planting nearly until harvest. This disease is favoured by wet and warm soil conditions, especially saturated conditions early in the growing season. In the early season, seedlings can be attacked and killed in the ground or soon after emergence. At seedling and later vegetative stages, infected stems appear bruised and are soft, secondary roots are rotted, the leaves turn yellow, and brown and plants can wilt and die.

During mid or late season, plants may die throughout the season. On infected plants, brown lesions form on the roots, the roots rot and degrade, and a dark chocolate-brown discoloration of the stem often extends from below the soil line upwards into lower parts of the plant. Leaves turn yellow, wilt and typically, stay attached after plant death. Plants are often killed in patches or in sections of rows [86]. Over the last 11 years, *Phytophthora* has suppressed US soyabean yields resulting in an estimated 500 million-bushel loss. Today, *P. sojae* remains a top threat for growers to manage worldwide and is the no. 1 disease that impacts soyabean yields [87].

Viruses

A number of viruses affect soyabean production on an annual basis (Table 6). Although yield loss from viruses is generally relatively low, individual fields may suffer from significant losses in any given year. Viruses are infectious submicroscopic particles made up of DNA or RNA enclosed by a protein coat. Soyabean viruses are generally transmitted by insect vectors such as aphids, beetles and whiteflies although Tobacco ringspot virus is also transmitted by the dagger nematode [88] (https://soybeans.ces.ncsu.edu/depts/pp/notes/Soybean/soy009/soy009.htm).

Depending on the genotype and age of infection, symptoms range from mosaic and mottling, leaf curling, green vein banding and stunting. Most severe symptoms are observed on plants infected at early growth stages.

Soyabean viruses with a worldwide distribution include the *Alfalfa mosaic virus* (*AMV*, *Alfamovirus*); Cucumber mosaic virus (*CMV*, *Cucumovirus*); *Soybean mosaic virus* (*SMV*, *Potyvirus*); *Tobacco streak virus* (*TSV*, *Ilarvirus*) and *Soybean yellow mosaic virus* (*SYMV*, *Potyvirus*) (Table 6). Viruses restricted to Asia are the *Indonesian soybean dwarf virus* (*ISDV*, *Luteovirus*); *mungbean yellow mosaic virus* (*MYMV*, *Geminivirus*) and the *soybean crinkle leaf virus* (*SCLV*, *Geminivirus*).

About 50 viruses have been reported to occur in soyabean in various parts of the world. Of these, 13 viruses have been reported from Southeast Asian countries. These viruses cause economic yield losses, and are sometimes a major limiting factor in soyabean production[89]. The most important soyabean viruses in Southeast Asia are *Soybean mosaic potyvirus* (*SMV*), *Peanut stripe potyvirus* (*PStV*), *Cowpea mild mottle Carlavirus* (*CMMV*), Mosaic *Cucumovirus*, Soybean Stunt Strain (*CMV-SS*), *Indonesian soybean dwarf Luteovirus* (*ISDV*), *Mungbean yellow mosaic Geminivirus* (*MYMV*), *Soybean crinkle leaf Geminivirus* (*SCLV*) and *Soybean yellow mosaic virus* (*SYM*). *SMV*, considered the most common virus found in soyabean throughout the world, is found in every Southeast Asian country where soyabean is grown. *PStV* is also widely distributed, since this virus is common in peanut plants throughout the region. Of the 13 soyabean viruses found in Southeast Asia, six are seed-borne and five are transmitted by vectors in a persistent manner [89].

Three most common soyabean virus diseases in northern Nigeria are the mosaic diseases, *cowpea mild mottle virus* (*CPMMV*, *Carlavirus*), transmitted by the whitefly, *B. tabaci*; *bean pod mottle virus* (*BPMV*, *Comovirus*); *alfalfa mosaic virus* (*AMV*, *Alfamovirus*); *cucumber mosaic virus* (*CMV*, *Cucumovirus*) and the *southern bean mosaic virus* (*SBMV*, *Sobemovirus*) [90].

The *BPMV* and *SMV* are the most common viruses infecting soyabean in North Carolina, USA [88]. Symptoms of the two most common viruses, *BPMV* and *SMV* may overlap and soyabean plants may be infected with both viruses. Tests to accurately identify each virus are available, but are rarely used, because taking corrective action is rarely possible. Tobacco ringspot virus (*TRSV*) is common in North Carolina but yield losses in production fields are generally insignificant.

***Bean pod mottle virus*:** *BPMV*, though generally less common than SMV can severely lower soyabean yield in infected fields. *BPMV* causes mottling of the leaves, leaf distortion, stunting of the plant, and mottled seed. *BPMV* has also been implicated in the 'green stem syndrome' and this can result in additional yield losses. Green stem syndrome is the condition where the stems of mature plants remain green and leathery making harvest difficult. The severity of symptoms is related to the virus strain, soyabean variety and how early the plant is infected. Yield loss to *BPMV* is generally related to time of infection; early infection can result in severe losses. Virus transmission through seed is very low, generally <0.01%. Like *SMV*, high temperatures limit *BPMV* symptom expression, whereas cool temperatures enhance development of leaf symptoms. BPMV is transmitted by several species of leaf feeding beetles, including bean leaf beetle, *Ceratoma trifurcata*. Overwintering beetles probably acquire virus from wild legume weed species. The incidence of BPMV infection in soyabean fields can be very high in years when overwintering bean leaf beetle populations are high [88].

***Soybean mosaic virus*:** *SMV* may be seed-borne or transmitted by aphids including the soybean aphid, *Aphis glycines*. *SMV* causes raised areas or puckering on the leaf surface, stunting of the plant and mottling of the seed. Seed mottling, however, may be caused by other factors. The primary leaves of plants grown from infected seed may show mottling and downwards curling. Infected plants are usually stunted because of shortening of the petioles and internodes, particularly when infected while still young or when infected from the seed. Some cultivars develop progressive necrosis of the petioles and stems, bud necrosis, defoliation and plant death with some strains resembling the bud blight of soyabean caused by Tobacco ringspot virus and Tobacco streak virus [91].

The pods are reduced in number and size, some are malformed, glabrous or seedless, and the yield may be considerably reduced. The virus can reduce the size of the seed, particularly in the case of co-infection with BPMV, change its chemical composition, diminish seed viability, seed germination and seedling vigour and thus the quality of the seed [92].

The severity of symptoms is related to the virus strain, soyabean variety and the age of the plant when it is infected. Symptom expression is also influenced by temperature. High temperatures limit symptom expression, whereas cool temperatures enhance development of leaf symptoms. Frequently, the crop may appear healthy until several days of cool weather in late summer or early autumn, when the entire crop appears to be affected [88].

Yield loss to *SMV* is generally related to time of infection. Virus transmission through seed may be as high as 30%. When infected seed is planted aphids may spread the disease from infected plants to the remainder of the soyabean crop. Wild hosts of *SMV* are relatively rare [88].

The incidence of *SMV* infection in USA soyabean fields has been much lower than that reported for *BPMV*. However, yield losses from SMV infection in the North Central Region have been as high as 94%. When soyabean plants are infected with both *BPMV* and *SMV*, symptoms can be more severe than infection by either virus alone [93].

***Tobacco ringspot virus*:** *TRSV* is found in all regions except in Europe (Table 6). Principal symptoms of *TRV* include stunting, leaf distortion and characteristic browning and curling of the terminal branch. The most obvious symptoms are the proliferation of buds and flowers, and lack of pods or poorly formed pods. Stems remain green and petioles may remain attached with black lesions. The stems remain green because the plants are sterile as a result of pod abortion. Infected plants will stand out in a mature soyabean field because of the green stems. If there are large numbers of these green plants, harvest may be difficult. The primary impact of this disease is that soyabean grown for seed from fields with this virus cannot be shipped to certain countries.

Although the virus may be seed-borne, this is probably not an important means of transmission since most infected plants are sterile. The dagger nematode can transmit this virus, but the virus does not move from the roots to the shoots and leaves of the soyabean plant [88]. Spread is probably by thrips and aphids, though no efficient vector has been identified [93].

***Tobacco streak virus*:** *TSV* has a worldwide distribution (Table 6) and a wide range of hosts [93, 94]. Plants of *Melilotus albus* (honey clover), a biennial legume, are the primary reservoir of *TSV* in the northwest USA [95]. Tobacco streak virus is transmitted by thrips, and through infected soyabean seed. TSV infection can be difficult to detect because the concentration of TSV particles decline significantly as soyabean plants mature [93].

TSV is prevalent in southeastern Brazil, and can limit yields of soyabeans in some years [96]. Epidemics of soyabean bud blight, caused by *TSV*, were monitored in two soyabean cultivars sown on seven different dates in 1987–88 in Arapoti County, Parana. The highest final level of disease incidence was 60% in 1987 and 90% in 1988 [97]. Brazilian bud blight is also a serious disease in the Santa Fe Province of Argentina [98]. It occurs in the USA [95, 99] but is of no economic significance on soyabeans because conditions are not conducive to disease development.

Nematodes

Plant-parasitic nematodes are recognized as one of the greatest threats to crops throughout the world. There are many nematode genera and species attacking soyabeans worldwide (Table 7).

As of 2015, 18 plant-parasitic nematode genera and 48 species have been associated with soyabean in South Africa alone [100]. Globally, the economically most important genera consist of *Ditylenchus* (peanut pod), *Heterodera* (cyst), *Hoplolaimus* (lance), *Meloidogyne* (root-knot), *Pratylenchus* (root-lesion), *Rotylenchulus* (reniform), *Scutellonema* (spiral), *Helicotylenchus* (spiral) and *Belonolaimus* (sting) (Table 7).

The root-knot nematodes, *Meloidogyne arenaria*, *M. hapla and M. javanica*; root-lesion nematodes (RLN), *Pratylenchus brachyurus*, *P. crenatus and P. thornei*; reniform nematode, *R. reniformis*; British spiral nematode *Scutellonema brachyurus*; common spiral nematode, *Helicotylenchus dihystera*; and the spiral nematode

H. pseudorobustus are present in all global regions (Table 7). The SCN, *H. glycines* is present in all regions except in Australia. The peanut pod nematode, *D. angustus*, occurs only in Africa, lesion nematode *P. scribneri* in North America, spiral nematode *S. truncatum* in Africa and the sting nematode *B. longicaudatus* in North America (Table 7).

Plant-parasitic nematodes are among the most serious disease problems in soyabean production. Disease caused by nematodes results in an estimated annual loss of 3–4% in soyabean yield the USA. A number of physical and biological factors influence the extent of damage caused by nematodes. Plant growth and yield are directly related to the numbers of plant-parasitic nematodes in the soil at planting (preplant population density). Most methods for managing these parasites involve lowering their initial numbers to levels at which plant damage is minimized. Soil type and texture have a marked influence on the damage caused by these organisms. Sandy soils favour most species of plant-parasitic nematodes. Since sandier fields have poor soil moisture-holding capacities and limited nutrient reserves, damage in these soils may be more severe. Soil pH and fertility problems as well as a hard pan tend to enhance damage caused by parasitic nematodes. Alleviating these problems will not eliminate nematode problems, but may be helpful [101].

SCN *H. glycines*: The SCN occurs in most soyabean growing regions [7] (Table 7). The nematode presents a threat to all regions of the world where soyabeans are grown and steps should be taken to prevent introduction in the first instance and to control spread once the nematode is known to be present [102]. The nematode is spread most easily via infested soil and contaminated machinery. Any mechanism that spreads infested soil can be a means of dispersal, including wind, water, migratory birds and seed lots [103].

H. glycines attacks a wide range of Fabaceae. Members of Caryophyllaceae and Scrophulariaceae are also hosts. Riggs and Wrather [104] provide a list of non-fabaceous hosts comprising 63 species in 50 genera from 22 families.

Symptoms on the root system range from slight discoloration to severe necrosis [2]. Diagnosis is confirmed when white or yellow females are observed attached to roots. Above ground symptoms, often not readily apparent, include slight to severe plant stunting and leaf chlorosis. Symptoms may be enhanced or repressed in association with other pathogens [105].

It is the most important soyabean disease in the USA [106]. It is particularly important in the semi-arid areas. Soyabean disease loss estimates were compiled for the 1994-harvested crop from the ten countries with the greatest soyabean production (Argentina, Bolivia, Brazil, Canada, China, India, Indonesia, Italy, Paraguay and the USA). The objective was to document the major soyabean disease problems in these countries and any recent changes in the severity of individual soyabean diseases. Total yield losses caused by *H. glycines*, in these ten countries, were greater than those caused by any other disease [107].

Roots infected with SCN usually have reduced Rhizobium nodulation and are short, sparse and dark in colour. With careful examination, small period-sized dot-like cysts on the roots can be seen. These are the developing females of the SCN and can range in colour from white to creamy yellow to dark brown [108].

Reported yield losses on soyabean vary from 10 to 70% in Japan [109, 110]. All soyabean growing areas in the USA are at risk. Losses in the southeastern USA were estimated at US$88.4 million in 1990 [111]. Wrather et al. [106] reported on losses due to *H. glycines* and other diseases on soyabean in the USA. *The* reduction in yield in 2002 amounted to US$784 million and yield losses were estimated at 3.6 million tonnes in 2001.

Columbia lance nematode: *Hoplolaimus columbus* has been reported from several different countries around the world, including infestations in India, Pakistan, Egypt and the United States [112]. In the USA, *H. columbus* is limited mainly to the southeastern part of the country. In sandy loam soils, on favourable hosts, it often replaces *Meloidogyne incognita* as the predominant species [113]. Maize, cotton and soyabean are good hosts for this nematode.

The Columbia lance nematode feeds both externally and internally on soyabean roots. Lesions may develop on the roots that can coalesce and give the appearance of a root rot [114]. Symptoms of feeding are taproot stunting, increased secondary branching, above ground stunting, wilting and mild chlorosis. Infected soyabeans bear only a few pods [113].

This nematode remains wormlike throughout its life cycle and can only be identified microscopically. The population densities of this nematode fluctuate only slightly during the course of a year. The amount of damage to soyabean and subsequent yield loss will be directly proportional to the density of this nematode at soyabean planting. Nematode densities are generally in the moderate-to-high range following these crops [114].

Root-knot nematodes, *Meloidogyne* spp.: Five root knot nematodes are of major importance; *M. arenaria*, *M. ethiopica*, *M. hapla*, *M. incognita* and *M. javanica*. Root-knot nematodes are generally the predominant plant parasitic nematodes associated with soyabean crops in South Africa [115, 116] and are considered as a serious constraint to production of the crop worldwide [117, 118].

Distinct root galling, caused by the feeding of female root-knot nematodes, represent below ground symptoms. Above ground symptoms, in fields where high population levels of root-knot nematodes occur, can include stunted plants and leaf yellowing [100].

M. incognita and *M. javanica* are the predominant nematode pests in South Africa [100] and are of major importance worldwide. *M. incognita* is predominantly found in warmer climates, at latitudes between 35°S and 35°N [119]. It is a damaging pathogen of soyabean and other crops throughout the southern United States [120]. In the 1970s, soyabean yield reductions reached as high as 90% for susceptible soyabean cultivars in the presence of *M. incognita* in some areas [121]. The impact of *M. incognita* on soyabean production has been reduced with the availability of resistant cultivars. However, annual soyabean yield losses to this nematode during 1999–2002 still exceeded 99.6 thousand metric tonnes in the USA [106].

RLNs, *Pratylenchus* spp.: Several species of RLNs may affect soyabeans globally (Table 7). However, the principal species are *P. brachyurus* and *P. crenatus*, which occur in all regions; *P. scribneri* which only occurs in North America; *P. teres*, a pest limited to Africa and *P. thornei* and *P. zeae* which occur in Africa, North America and Australia.

P. brachyurus has more than 80 hosts worldwide [122]. *P. brachyurus* is the principal soyabean nematode in Southeastern USA. This nematode, like Columbia lance nematode, remains wormlike throughout its life cycle. Lesion nematodes penetrate soyabean roots, feed internally on the root, and lay eggs within the root system [101]. The symptoms caused by this parasite are non-specific. Damage symptoms are reduced root systems with necrotic streaks or lesions. The lesions on roots may coalesce to give the appearance of a root-rot [101]. Leaves wilt and have abnormal colours, stems show rosetting and seeds have lesions. Severe damage kills entire plants [122].

Damage to soyabean caused by this pathogen is usually restricted to sandy loam and sandy soils in the southeastern coastal plain of North Carolina. Delaying soyabean planting until mid-June, as done with double-crop wheat/soyabean systems or rotation with maize, frequently can be used to manage this nematode. Tobacco and peanuts should not be used in rotation with soyabean if lesion nematode is a problem. While these crops are poor hosts for lesion nematode, they may be damaged by the high numbers that build up on soyabean [101].

Pratylenchus thornei is one of the most widely distributed species of *Pratylenchus* and has been reported from every continent except from Antarctica [123]. A major host is wheat but it has about 40 other hosts including soyabean [122]. Of the 22 predominant nematode species recovered from soyabean soil in South Africa, *P. thornei* ranks 9th in mean population density compared with the most abundant species *P. zeae* [116].

It is an obligate migratory endoparasite that first feeds externally then enters plant roots, feeds, reproduces and moves freely within the tissue while spending its entire life cycle there. The species can also be found in soil around roots. Within the roots, feeding is confined to the root cortex. Like other *Pratylenchus* species, *P. thornei* has six life stages: egg, four juvenile stages and adults. Reproduction is by parthenogenesis [124].

In general, root lesion infection results in plants exhibiting symptoms of chlorosis, wilting and stunting. Infected roots show initial symptoms of small, water-soaked lesions that soon turn brown to black. Lesions are formed along the root axis and may coalesce laterally to girdle the roots that are killed. Affected root tissue may slough off leaving a severely reduced root system. Secondary infection by fungi and bacteria may further destroy the root system by causing the sloughing off the root tissues and rot. Plant yield is reduced and in severe infections, plants may be killed [124].

Reniform nematodes *Rotylenchulus* spp.: Reniform nematodes in the genus *Rotylenchulus* are semi-end parasitic (partially inside roots) species in which the females penetrate the root cortex, establish a permanent-feeding site in the stele region of the root and become sedentary or immobile. The anterior portion (head region) of the body remains embedded in the root whereas the posterior portion (tail region) protrudes from the root surface and swells during maturation. The term ‘reform’ refers to the kidney shaped body of the mature female [125].

Rotylenchulus reniformis is largely distributed (Table 7) in tropical, subtropical and warm temperate zones in South America, North America, the Caribbean Basin, Africa, southern Europe, the Middle East, Asia, Australia and the Pacific [126]. It was first found on cowpea roots in Hawaii [127].

At least 314 plant species can act as hosts to reniform nematodes. Among them, cotton, cowpea, soyabean, pineapple, fruit crops, tobacco tea and various vegetables are the most common hosts [101]. Robinson *et al.* published a list of hosts and nonhosts for the reniform nematode [128].

Only females infect plant roots. After infection, a feeding site composed of syncytial cells is formed. A syncytial cell is a multinucleated cell resulting from cell wall dissolution of several surrounding cells.

R. reniformis has a life cycle similar to those of root knot and cyst, but signs or symptoms for the reniform nematode are not readily visible to the naked eye. The amount and type of damage incurred often depends on the host species and/or cultivar as well as the nematode population. General symptoms include reduced root systems, leaf chlorosis, overall stunting of host plants, and reduced yields and plant longevity [125].

Spiral nematodes

Sting nematode, *Belonolaimus longicaudatus*: Sting nematodes are endemic to the southeastern United States (Table 7) and are only capable of reproducing in sandy soils. This fact is the primary limiting factor in this nematodes distribution. It is an extremely damaging nematode to cotton, maize, peanut and soyabean [101]. This pathogen is always wormlike and microscopic. The sting nematode is an ectoparasite that feeds externally on the root and lays its eggs in the soil.

Sting nematodes do not enter plant roots to feed. They inject a toxic enzyme into the roots of their host while feeding, resulting in significant damage, yield loss and even plant death [129]. Symptoms of sting nematode damage are poor growth and short roots with many rootlets at the ends of the roots. Roots may have a ‘stubby’ appearance. Plants that escape damage from this nematode early in the season may have a taproot with a few lateral roots, but no fibrous root system. Fortunately, the occurrence of this nematode is limited to very sandy soils. Fields infested with this nematode should be considered for uses other than row crops. Watermelon and tobacco will not support reproduction of this nematode and can be used in rotation with soyabean where this nematode is a problem [101].

Weeds

There are numerous weed species that are important biotic constraints in soyabean production worldwide of which the key species are listed in Table 8. The weed species are not specific to soyabeans and species composition depends on the locally occurring species, which are general pests of most local crops.

Heavy weed infestations in soyabeans greatly interfere with the timeliness and efficiency of harvests, and if not controlled, cause severe losses in crop production. Losses can reach 100% depending on the soil preparation, status of the crop, weed species present and environmental factors. Studies in India indicated a 42% loss in soyabean yield when comparing yields in weedy check plots compared with weed free plots. Net returns, due to weed infestation, were reduced by 53% [130].

Perennial and most annual weeds are a problem in soyabean in the early growth stages. Because soyabeans are weak competitors in the early part of the crop season, weeds outgrow them. Soyabeans usually develop a full canopy by 8 weeks after emergence and can then compete effectively with weeds up to maturity. Little or no reduction in yield occurs if soyabean is kept weed free for the first 4 weeks after plant emergence. This is the critical period for weed competition in soyabeans [131]. Therefore, weed control at an appropriate time, using a suitable weed control method, is a requirement to ensure effective weed management and high grain yields in soyabean.

Morphologically weeds are classified as *Monocotyledons* (grasses and sedges) and *Dicotyledons* (broad-leaved). The top ten worst weeds in corn (maize) and soyabeans as summarized by US university scientists [132] list eight broad-leaved weeds and two grassy weeds. Broad-leaved weeds are: velvetleaf, *Abutilon theophrasti*; common water-hemp, *Amaranthus tuberculatus* and Palmer amaranth, *A. palmeri*; giant ragweed, *Ambrosia trifida*; common lambsquarters, *Chenopodium album*; horseweed/marestail, *Conyza canadensis*; bur cucumber, *Sicyos angulatus* and three-lobe morning glory, *Ipomoea triloba*. The two grassy weeds listed among the top ten worst weeds are: woolly cupgrass, *Eriochloa villosa* and giant foxtail, *Setaria faberi* (Table 8).

Broad-leaved weeds that have a worldwide distribution are slender amaranth, *A. viridis*; common lambsquarters, *C. album*; horseweed/marestail, *C. canadensis* and wild poinsettia, *Euphorbia heterophylla*. Grassy weeds that are present in all global regions are crowfoot grass, *Dactyloctenium aegyptium*; goose grass, *Eleusine indica*; yellow foxtail, *Setaria pumila* and green foxtail, *S. pumila* (Table 8).

Invasive weeds

Some of the most serious weeds in soyabeans are referred to an 'invasive' or 'alien' species. Due to the disturbance of the soil in soyabean crop production, many areas are infested with invasive weed species. The term 'invasive' as most often used applies to introduced species (also called 'non-indigenous' or 'non-native') that adversely affect the *habitats* and *bioregions* they invade economically, environmentally or ecologically.

Common invasive species traits include the following: fast growth, rapid *reproduction*, high *dispersal* ability, *phenotypic plasticity* (the ability to alter growth form to suit current conditions), tolerance of a wide range of environmental conditions (*ecological competence*), ability to live off of a wide range of food types (*generalist*), association with humans [133] and prior successful invasions [134].

Among the broad-leaved species, that are severe invasive weeds, are the pigweeds (Amaranthaceae), common waterhemp, *A. tuberculatus*; slender amaranth, *A. viridis*; and Palmer amaranth, *A. palmeri.* Serious invasive grassy weed species include the foxtail (Poaceae) species, giant foxtail, *S. faberi*; yellow foxtail, *S. pumila* and green foxtail, *S. viridis* (Table 8).

Pigweeds

Amaranthus, collectively known as amaranth [135], is a cosmopolitan genus of annual or short-lived perennial plants. Some amaranth species are cultivated as leafy vegetables, pseudocereals and ornamental plants. Most of the *Amaranthus* species are summer annual weeds and are commonly referred to as pigweed.

Pigweed is the common name for several closely related summer annuals that have become major weeds of vegetable and row crops throughout the United States and much of the world [136]. Most pigweeds are tall, erect-to-bushy plants with simple, oval- to diamond-shaped, alternate leaves and dense inflorescences (flower clusters) comprised of many small, greenish flowers.

Pigweeds thrive in hot weather, tolerate drought, respond to high levels of available nutrients and are adapted to avoid shading through rapid stem elongation. They compete aggressively against warm season crops, and reproduce by prolific seed production.

Also called amaranths, pigweeds are native to parts of North and Central America. Crop cultivation and human commerce have opened new niches, allowing pigweeds to invade agricultural ecosystems throughout the Americas, and parts of Europe, Asia, Africa and Australia (Table 8). Most amaranths make nutritious green vegetables or grain crops, and deliberate planting for food has helped some weedy species spread around the world [136].

Pigweeds are highly responsive to nutrients, especially the nitrate form of nitrogen (N) [137]. Fertilization enhances both weed biomass and seed production.

Table 8. Pests of soyabean and their geographical distribution: weeds

Weeds	Asia	Africa	Europe	N. America	S. America	Australia
Broad-leaved weeds						
Velvetleaf	X	X	X	X		
A. theophrasti						
Common waterhemp						
A. tuberculatus				X		
Slender amaranth						
A. viridis	X	X	X	X	X	X
Palmer amaranth						
A. palmeri			X	X		X
Giant ragweed						
A. trifida	X		X	X		
Common lambsquarters						
C. album	X	X	X	X	X	X
Horseweed/marestail						
C. canadensis	X	X	X	X	X	X
Burcucumber						
S. angulatus	X		X	X		
Three-lobe morning glory						
I. triloba	X	X		X	X	X
Asian spider flower						
Cleome viscosa	X					
Wild poinsettia						
E. heterophylla	X	X	X	X	X	X
Common sunflower						
Helianthus annuus				X		
Grassy weeds						
Crowfoot grass						
D. aegyptium	X	X	X	X	X	X
Goose grass						
E. indica	X	X	X	X	X	X
Woolly cupgrass						
E. villosa	X			X		
Giant foxtail						
S. faberi	X			X		
Yellow foxtail						
S. pumila	X	X	X	X	X	X
Green foxtail						
S. viridis	X	X	X	X	X	X
Horse purslane						
Trianthema portulacastrum	X					

Pigweeds in soyabeans are reported to host pest nematodes (*Meloidogyne* spp.) and many crop pathogens, including fungi that cause white mould (*S. sclerotiorum*) and southern blight (*Sclerotium rolfsii*) [137].

The pigweeds, common waterhemp *A. tuberculatus*, and slender amaranth and Palmer amaranth, *A. viridis* and *A. palmeri*, are included in the list of the top ten worst weeds in maize and soyabeans [132]. Common waterhemp is restricted to West Asia, Europe and North America [138] while slender amaranth [139] and Palmer amaranth occur worldwide (Table 8).

A. tuberculatus is an annual dioecious herb, 1–2 m tall, which has spread from its native range in northern North America and is considered a major weed of agricultural fields and other disturbed areas in 40 US states. In the mid-western USA, it has become increasingly difficult to control over the past 10 years due to a persistent seedbank and the development of resistance to certain herbicides. *A. tuberculatus* seed is a known contaminant of soyabean seed and other grains, and has been accidentally introduced and become naturalized in parts of West Asia and Europe. Potential spread to other areas is considered likely.

A distinguishing characteristic of *A. tuberculatus* that sets it apart from similar members of the genus *Amaranthus* is the lack of hair on its stems and leaves. This characteristic gives the plant a bright, glossy appearance *Amaranthus viridis*, an annual herb, which grows from 6 to 100 cm high [139], is native to the southern United States and Mexico [140]. It is one of the most common weeds in the tropics, subtropics and warm temperate regions. *A. viridis* occurs virtually in all disturbed habitats and all crops, herbaceous and woody, in all but the wettest soils throughout the warmer regions of the world. Other *Amaranthus* species may be confused with this plant but *A. viridis* can be distinguished by its wrinkled fruit and its flower bracts being <1 mm long and acute (pointed).

A. viridis is used as a traditional medicine in North America. It is eaten in the northeastern Indian state Manipur where it is known as *Cheng-kruk* and eaten traditionally as a vegetable in South India, especially in Kerala, where it is known as '*Kuppacheera*'. It is also eaten as a vegetable in parts of Africa [141]. In Jamaica it is eaten as a vegetable and is known locally as 'callaloo'. Its use as a food may be the reason that this invasive weed has spread globally.

Amaranthus palmeri is a species of edible flowering plant in the amaranth genus. It has several common names, including carelessweed, dioecious amaranth, Palmer's amaranth, Palmer amaranth and Palmer's pigweed. The leaves, stems and seeds of Palmer amaranth, like those of other amaranths, are edible and highly nutritious. Palmer amaranth was once widely cultivated and eaten by Native Americans across North America, both for its abundant seeds and as a cooked or dried green vegetable. It is native to most of the southern half of North America. Populations in the eastern United States are probably naturalized. It has also been introduced to Europe, Australia and other areas [142].

Palmer amaranth is the most competitive and aggressive pigweed species. Season-long competition by Palmer amaranth at 2.5 plants per foot of row can reduce soyabean yield by as much as 79% [143]. In Kansas, USA soyabean yield reduction of about 78% was reported by Palmer amaranth, at a density of 8 plants/m^2. Early emerging Palmer amaranth plants also cause more soyabean grain yield loss when compared with later-emerging plants [144]. Palmer amaranth, referred to as a 'superweed,' is considered a threat most specifically to the production of genetically modified (GM) cotton and soyabean crops in the southern United States, because it has developed resistance to glyphosate. It has also been referred to as 'a weed strong enough to stop combines' [145].

Palmer amaranth emerges later than many summer-annual weeds and continues to emerge throughout the growing season. This extended emergence pattern makes it difficult for preemergence and nonresidual postemergence herbicides to control later-emerging plants.

The widespread adoption of no-tillage systems, reduced reliance on soil-applied residual herbicides, and increased herbicide resistance have contributed towards the increased infestation of Palmer amaranth in different cropping systems [146].

The high relative growth rate of Palmer amaranth makes control with postemergence herbicides difficult. In the southern United States, Palmer amaranth has been documented to grow as much as 2.5 inches per day [143]. Unlike other pests, it can continue to grow an inch a day even without water, making it particularly adept during drought. It also thrives in hot weather, continuing to grow when temperatures top 90 °F and other plants shut down [147].

Each female plant produces as many as 500 000 seedlings, meaning just one plant can birth an entire field [147]. It can hybridize with other pigweeds. Its dioecious reproduction (separate male and female plants) forces outcrossing and genetic diversity that allows it to readily adapt to new environmental conditions and quickly spread herbicide resistance genes when selection pressure is applied [148].

Foxtails

The foxtails, members of the *Setaria* genus, are one of the worst weed groups interfering with North American and world agriculture and land management [149, 150]. The inflorescence resembles a foxtail, hence the common name. The weedy *Setaria* spp., *S. faberii* (giant foxtail), *S. pumila* (yellow foxtail) and *S. viridis* (green foxtail) [151, 152] are considered among the top ten worst weeds [132].

Five successive waves of *Setaria* spp. invasion from pre-agricultural times to the present have resulted in widespread infestation of the disturbed, arable, temperate regions of the earth. These invasions have resulted in considerable economic and environmental costs [132]. The origin of the genus and the original wild Eurasian *Setaria* sp. was probably Africa, based on the large number of *Setaria* spp. from there (74 of 125), as well as genomic evidence of tropical origins [152, 153].

The success of the *Setaria* spp.-group is due to their intimate evolutionary relationship with humans, disturbance, agriculture and land management. Genotypic and phenotypic biodiversity provide this species-group with traits that allow them to invade, colonize, adapt to and endure in a wide range of disturbed habitats in temperate, tropical and sub-tropical regions [154].

S. faberi, giant foxtail, also referred to as Japanese bristlegrass, is an annual grass, reproducing by seeds. It can reach 0.61–1.5 m in height. Leaves are up to 41 cm long, 15–25 mm wide. The leaf blade has short hairs that cover the upper surface. The ligule has a fringe of hairs [155]. Flowering occurs in late summer to early autumn, when a green (eventually straw-coloured), bristly inflorescence develops.

S. faberi, a native of Asia, was accidentally introduced into the United States in the 1920s as a contaminant of other grain [155]. It is also present in some European countries [156]. Plants invade disturbed sites such as fields, roadsides, landfills, fencerows and right of ways.

Giant foxtail is one of the most prevalent annual grasses infesting crop production fields in the Midwest, USA [157]. This is partially due to its prolific production of seed and their subsequent longevity in the soil. Giant foxtail interferes with soyabean growth and reduces yield. In a 3-year study, an infestation of 54 plants per 0.3 m row reduced soyabean seed yield 28%. In addition, the increase in giant foxtail dry matter with increasing weed density was proportional to the decrease in dry matter of the soyabeans.

S. pumila, yellow foxtail, is a cosmopolitan species that occurs in all regions (Table 8). Soyabeans are among the many hosts worldwide [158]. *S. pumila* is a tufted annual grass, very variable in size and vigour ranging from a few centimetres up to more than 100 cm high. It is much branched and there may be rooting at lower nodes. The stem base and lower leaves are often red or purplish, especially under infertile soil conditions. The leaves are generally glaucous ('gleaming', the meaning of 'pumila'), up to 30 cm long by 1 cm wide, and glabrous except for a few long hairs on the upper surface near the base [158].

S. pumila can act as an alternative host of *Claviceps purpurea* (ergot), *Pythium graminicola* (root rot), *Sclerospora graminicola* (downy mildew), *Ustilago neglecta* (smut), *Anoecia corni* (dogwood aphid), *Rhopalosiphum maidis* (green corn aphid), *Pyricularia grisea* (rice blast), *Cirphis compta* (paddy cutworm) and *Diabrotica longicornis* (northern corn rootworm).

Setaria viridis, a species of grass known by many common names, including green foxtail, occurs in all world regions [159] (Table 8). It is sometimes considered a subspecies of *Setaria italica*. It is native to Eurasia, but it is known on most continents as an introduced species and is closely related to *S. faberi*, a noxious weed. A hardy grass, it grows in many types of urban, cultivated and disturbed habitats. It is the wild antecedent of the crop foxtail millet [160].

S. viridis is a native of Europe but spread to North America as early as 1821 [161] and now occurs in most temperate countries of the Northern and Southern hemispheres. It rarely occurs in the tropics other than at high altitudes.

Management of Soyabean Pests

A detailed package of practices for managing soyabean pests, diseases and weeds has been developed in India [162]. The management practices are based on crop stages: period between crops, pre-sowing, sowing, vegetative stage, reproductive stage and harvest (Table 9). At each crop growth stage, the major pest management practices employed consist of (1) mechanical methods; (2) cultural controls; (3) host plant resistance; (4) biological control agents (parasitoids, predators and pathogens); (5) microbial control (biopesticides) (fungi, viruses, bacteria, nematodes); (6) botanical pesticides and (7) chemical control (synthetic organic pesticides) [163, 164].

Mechanical methods refer to the reduction or suppression of insect populations by means of manual devices. Some mechanical methods are hand picking, clipping, pruning and crushing to destroy insects and roguing of disease affected seedlings. Although, mechanical methods are labour intensive they may be economical if labour is abundant and not expensive. Mechanical practices at the vegetative stage are the collection and destruction of girdle beetle infested plant parts and egg masses and the roguing of *Seclerotium* affected seedlings and yellow mosaic virus affected soyabean plants (Table 9).

Cultural control practices include all of the crop production and management techniques that are utilized by farmers to maximize their crop productivity and/or farm income [163]. Cultural measures involve the manipulation of the agroecosystem to make it unfavourable for insect pests and more suitable for crop growth and natural enemies of the pests. Cultural practices have potential as components of IPM for farmers in the tropics because they are simple, effective, convenient, environmentally safe, socially acceptable and economically feasible and do not require any limitation posed by pesticides to the environment, human health and development of pest resistance [165].

Numerous cultural control practices are utilized in managing soyabean pests, diseases and weeds (Table 9). Practices used in the stage *between soyabean crops* are crop rotations, keeping soils covered with living vegetation, adding organic matter in the form of farmyard manure (FYM), vermicompost and decomposed crop residue, reduced tillage intensity and removal of infected stubbles followed by deep summer ploughing. *Pre-sowing* cultural practices include the application of biofertilizers and soil nutrients such as NPK and S and selection of clean, disease and weed free seeds of high yielding insect pest and disease resistant/tolerant varieties. Cultural practices applied at the time of *sowing* include sowing a trap crop, sowing time, seed rate and plant spacing. At the *vegetative stage* of soyabean, maintaining a weed-free crop for 30–45 days by hand weeding, is an effective cultural control of weeds. At harvest time, the crop should be harvested when 85% of the pods have turned brown for a shattering variety and 80% for a non-shattering variety. Late harvesting, under humid conditions, may result in the development of *Phomopsis*, a fungal pathogen that results in reduced seed germination.

Host plant resistance as first described by Painter [166] is 'the relative amount of heritable qualities that influence the ultimate degree of damage done by an insect'. In practical agriculture, resistance represents the ability of a certain variety to produce a larger crop of good quality than do ordinary varieties at the same level of insect population. This definition originally referred to insects, but has since been broadened to include plant resistance to diseases.

Globally, numerous soyabean varieties have been bred for resistances/tolerance to soyabean insects and diseases. Based on previous knowledge of the most important pests, diseases and nematodes, in a given location, seeds of resistant varieties can be selected in the pre-sowing phase. Table 9 provides a list of conventionally bred soyabean varieties with resistance/tolerance to four important insect pests and 12 important fungal, bacterial and viral diseases in India.

GM soyabeans were first introduced in 1996 [167], principally to make soyabean crops resistant to herbicides. Although resisted in some regions, notably Europe, GM soyabeans are now grown in many parts of the world. Much of the soyabean grown in Latin America and the USA (>90%) is GM to tolerate glyphosate herbicide. This means that soyabeans can be sprayed several times with this herbicide during the growing season and all other plants,

Table 9. Soyabean pest management package (modified from [90, 163])

Crop stage	Management tactic		Purpose
Between crops			
	Cultural control		
	Crop rotations with cereal crops		Break the continuity of soil-borne pests as well as attract beneficial insects and predatory birds
	Add organic matter in the form of FYM, vermicompost, decomposed crop residue		To enhance below ground biodiversity
	Reduce tillage intensity		Conserve hibernating natural enemies
	The removal or destruction of stubble and crop residues by burning, flooding or ploughing after the crop has been harvested		Removes disease- and insect-infected plants and exposes soil-borne pathogens, nematodes and insect-pests, and rhizomes and bulbs of perennial weeds
Pre-sowing			
Field preparation	*Cultural control*		
	Pre-sowing tillage		Reduces the potential for carryover of pest populations from one season to another by exposing larvae to desiccation and predation
	Apply balanced dose of biofertilizers and nutrients at recommended rates as per the soil test		To promote good plant growth and compete with weeds
	Biological control		
	Apply *Trichoderma*/*P. fluorescens* as a soil application		For the management of seed, seedling and seed-borne foliar diseases
	Apply mycorrhiza and PGPR		Promote plant growth and antagonism to soil-borne pathogens
	Host plant resistance		
Seed selection	Selection of high yielding insect pest and disease resistant/tolerant varieties mentioned below:		Insect and disease control
	Insect pest	Resistant/tolerant varieties	
	Stem fly	JS 335, PK 262, NRC 12, NRC 37, MACS 124 and MAUS 2, MAUS 47	
	Tobacco caterpillar	JS 81-21, PS 564 and PK 472	
	Green semilooper	NRC 7, NRC 37, PUSA 16, PUSA 20, PUSA 24, JS 93-05, JS 97-52, MAUS 47 and JS 80-21	
	Girdle beetle	JS 71-05, NRC 7, JS 97-52, MAUS 32 and Indira Soya 9	
	Disease	Resistant/tolerant varieties	
	Rust	PK 1024, PK 1029, JS 80-21, Indira soyabean 9 and MAUS 61-2	
	Sclerotium blight	NRC 37	
	Charcoal rot	NRC 2, NRC 37, JS 71 05, LSb 1, MACS 13 and JS 97-52	
	Rhizoctonia aerial blight	PK 472, PK 1042, PK 564 and SL 295	
	Anthracnose and pod blight	Bragg, Himso 1563, PK 472, JS 80-21, Pusa 37, VLS 21 and NRC 12	
	Bacterial pustule	PK 1029, PK 1042, JS 71-05, JS 90-41, Bragg, Himso 1563, Indira soya 9, KHSb 2, MAUS 32, NRC 7, NRC 37 and VLS 2	
	Yellow mosaic virus	PK 416, PK 472, PS 564, PK 1024, PK 1029, PK 1042, Pusa 37, SL 295, SL 525, SL 688 and JS 97 52	

Table 9. (Continued)

Crop stage	Management tactic		Purpose
	Soybean mosaic virus	JS 71–05, KHSb 2, LSb 1, MACS 58, MACS 124, Punjab 1 and VLS 2	
	Myrothecium leaf spot	JS 71-05, JS 335, MACS 13, MACS 124, MAUS 47 and NRC 7	
	Purple seed stain	JS 80-21 and Bragg	
	Frog eye leaf spot	Bragg, JS 80-21, KHSb 2 and VLS 21	
	Alternaria leaf spot	KHSb 2, NRC 2, PK 327, PK 1042, Himso 1563, JS 80-21, Pusa 37 and VLS 21	
	Cultural control		
Seed cleaning	Clean seeds to be sure they are free from disease infestation, and weed seeds		
Seed germination	Test seeds for germination before planting. The germination rate should be 85% or more		To avoid transferring diseases and weeds to the newly planted crop
	Biological + chemical control + cultural control		A good crop stand competes with weeds
Seed treatment	Apply *Trichoderma viride* at 5 g or thiram 37.5% + carboxin 37.5% DS at 3 g/kg seed		For the management of seed, seedling and seed-borne foliar diseases
	This should be followed by seed treatment with *Bradyrhizobium* and phosphate solubilizing bacteria (PSB) at 5 + 5 g/kg seed		*Bradyrhizobium* fixes nitrogen and PSB increases P uptake by the plant, both causing vigorous plant growth, which results in tolerance to insect attack and competition with weeds
Sowing			
	Cultural control		
Sowing a trap crop	Use of castor and dhaincha (green manure) as a trap crop		Control of tobacco caterpillar and girdle beetle respectively
	Early planting of border strips as a trap crop		Attract and control stink bugs and bean leaf beetle
Sowing time	Cultivation of soyabean in rainy season only and sowing should be done timely when soil moisture is sufficient (8–12 cm depth)		To ensure proper germination and plant growth to compete with weeds
Seed rate	Optimum seed rate (65–75 kg/ha) should be used depending upon seed size. After every 15 rows, a gap of one row should be given		To provide proper plant growth and provide moving space for spraying in standing crop
	Inter-cropping soyabean either with an early maturing variety of Devil's dung (asafoetida) (*Ferula assa-foetida*) or maize or sorghum in the sequence of four rows of soyabean with two rows of intercrop		Repel pests and promotes biodiversity which will help to increase and conserve natural biocontrol fauna e.g. coccinellid beetles, *Chrysoperla* etc. In girdle beetle and semilooper endemic areas, intercropping with maize or sorghum should be avoided
Row spacing	Select proper row spacing as locally recommended		Affect oviposition behaviour of lepidopterous pests
Plant spacing	Sow soyabean by hand, planter, or by drilling. Plant three to four seeds/hole at a spacing of 75 cm between rows and 10 cm between stands		Obtain maximum high-quality yields and limit the population and damage of insect pests by modifying the environment and influencing the effectiveness of natural enemies
Vegetative stage			
	Mechanical practices		
	Collection and destruction of girdle beetle infested plant parts, egg masses		Control insect pests
	Roguing of *Sclerotium* affected seedlings and yellow mosaic affected plants		To prevent soyabean diseases from spreading
	Biological control		
	Erection of bird perches at 10–12/ha		Increase bird predation of insect pests

Table 9. (Continued)

Crop stage	Management tactic	Purpose
	Conserve spiders, coccinellid beetles, tachinid fly, praying mantids, dragon fly, damsel fly, *Chrysoperla* and meadow grass hoppers through biopesticides or minimum use of broad spectrum pesticides	To exploit maximum potential of bio-control fauna
	Release *T. remus* at 50 000/ha	To control *S. litura* via egg parasitization
	Microbial control	
	Spray *B. thuringiensis* var. *kurstaki*, serotype H-39, 3b, strain Z-52 at 0.75–1.0 kg/ha	For management of the semilooper complex (*C. acuta*, *Gessonia gemma*, *D. orichalcea*) and other defoliators
	Botanical pesticides	
	Spray of NSKE at 5%	For control of early stage larvae and sucking pests
	Chemical control	
	Application of pesticides should only be resorted if alternative practices are not effective and pest population crosses the economic threshold	Management of insect pests and soyabean diseases
	Cultural control	
	Maintain optimum and healthy crop stand	For the soyabeans to compete with weeds
	The crop should be maintained weed free initially for 30–45 days by resorting two hand weedings	To prevent weed completion with soyabeans
Reproductive stage		
Flowering	*Chemical control*	
	Apply triazophos 40% EC at 625 ml/ha or chlorantraniliprole 18.5% SC at 150 ml/ha	To control tobacco caterpillars, stem fly and girdle beetle
Pod development	Poison baiting with 2% zinc phosphide at podding and green seed stage preceded by 1 day pre-baiting or application of bromadiolone 0.005%	Rodent control
Harvest		
Time	*Cultural control*	
	Harvest when 85% of the pods have turned brown for a shattering variety and 80% for a non-shattering variety	To avoid development of seed-borne diseases e.g. *Phomopsis* which reduce seed germination

but the soyabeans, will be killed. Recently more and more weeds have become resistant to this herbicide, and therefore, new GM soyabean varieties have been developed with multiple herbicide resistance. As of 2017, 0% of the soyabeans grown in India are GM.

Biological control as defined by Van Driesche and Bellows (1996) [168] is 'the use of parasitoids, predators, pathogens and antagonists or competitor populations to suppress a pest population, making it less abundant and less damaging than it would otherwise be'. Insects, mites, weeds, plant diseases and vertebrates all may be targets of biological control. Biological control may be the result of purposeful action by man or may result from the unassisted action of natural forces. Biological control may be employed either for suppression of crop pests or for restoration of natural systems affected by non-native (invasive) pests.

As practiced, biological control can be self-sustaining and distinguishes itself from all other forms of pest control by acting in a density dependent manner [169]. In a strict ecological sense, applied biological control can be considered a strategy to restore functional biodiversity in agroecosystems by adding, through classical and augmentative biocontrol techniques, 'missing' entomophagous insects or by enhancing naturally occurring predators and parasitoids through conservation and habitat management [170].

In the Indian soyabean pest management package at *Pre-sowing* it is recommended that *Trichoderma or Pseudomonas fluorescens*, mycorrhizae and plant growth promoting rhizobacteria (PGPR) be applied to the soil at time of field preparation for the management of soil-borne diseases (Table 9). These fungal and bacterial antagonists are used to promote plant growth and provide the control of plant diseases.

Trichoderma is the most widely used biocontrol agent [171]. Among the various isolates of *Trichoderma*, *T. viride*, *T. asperellum*, *T. harzianum*, *T. virens* and *T. hamatum* are used for the management of various diseases of crop plants including the *Fusarium*, *Phytophthora* and *Scelerotia* genera. **Q8** *Trichoderma* has many advantages including (a) highly competitive saprophytic ability, (b) enhances plant growth, (c) produces secondary metabolites/antibiotics of volatile and non-volatile nature, (d) provides a broad spectrum of action against various pathogens, (e) provides excellent and reliable control, (e) is environmentally safe and (f) is easy to multiply, even at the farm level [171, 172].

Mycorrhizal fungi form a mutualistic relationship with the roots of most plant species. In such a relationship, both the plants themselves and those parts of the roots that host the fungi are said to be mycorrhizal. Mycorrhizal plants are often more resistant to diseases, such as those caused by microbial soil-borne pathogens.

Bacteria that colonize plant roots and promote plant growth are referred to as PGPR. PGPR are highly diverse. Their effects can occur via local antagonism to soil-borne pathogens or by induction of systemic resistance against pathogens throughout the entire plant [173].

Three biological control activities are listed at the *Vegetative* stage (Table 9). (1) Erection of bird perches in the field increases the number of birds which are predators of soyabean insect pests. (2) To exploit the maximum potential of biocontrol fauna the conservation of natural enemies (spiders etc.) of pest insects through the application use of biopesticides or the minimum use of broad spectrum of pesticides is recommended. (3) The release of the egg parasitoid, *Telenomus remus* is recommended to control the armyworm, *Spodoptera litura*.

Microbial control refers to the exploitation of disease causing organisms to reduce the population of insect pests below damaging levels. Microbial agents, also referred to as biopesticides, that provide pest control include the viruses, bacteria, fungi and nematodes. Sprays of the bacterium, *Bacillus thuringiensis* var. *kurstaki*, are recommended for the management of the semi-looper and other defoliators at the *Vegetative* stage in India (Table 9). Because biopesticides are specific to the target pests, their natural enemies are not affected. Some other biopesticides include the nuclear polyhedrosis virus (NPV), the fungi *Metarhizium anisopliae* and *Metarhizium* (*Nomuraea*) *rileyi* and entomopathogenic nematodes belonging to the families Heterorhabditidae and Steinernematidae.

Botanical pesticides are agricultural pest management agents whose active ingredients is are plant-produced chemicals. One of the most widely used botanical pesticides is produced by the neem tree, *Azadirachta indica*, and is commonly referred to as 'neem'. Spray application of Neem Seed Kernel Extract (NSKE) is recommended in India for the management of early stage insect pest larvae and sucking pests (Table 9).

The neem tree is indigenous to India from where it has spread to many Asian and African countries. For centuries, the tree has been held in esteem by Indian folk because of its medicinal and insecticidal value. All parts of the neem tree possess insecticidal activity but seed kernel is the most active. Effects on insect pests have been reported to be repellency, antifeeding, growth inhibition, development defects and even mortality [163].

Neem products are safe to spiders, adults of numerous beneficial insect species and eggs of many predators such as coccinellids. Nymphal/larval instars are more or less susceptible, especially under laboratory conditions. As a rule, no or only slight side effects, are observed under semi-field or field conditions. Due to their relative selectivity ('stage selectivity'), neem products can be recommended for many IPM programmes as it is unlikely that they will cause severe disturbances in ecosystems [174].

Chemical control (synthetic organic pesticides), because of their environmental hazards, are only recommended as a last resort, when other control measures in an IPM programme are not effective. In the soyabean package of practices, pesticides are recommended at the *Vegetative* stage for the control of insect pests and diseases (Table 9). In the *reproductive* stage, at time of flowering, triazophos (organophosphate) or chlorantraniliprole (anthracitic diamine, a reduced risk insecticide) are recommended for control of stem fly, tobacco caterpillar, green semilooper and the girdle beetle.

Soyabean pest management under a changing climate

In recent decades, climate change and the resultant global warming has become an issue of serious concern worldwide for existence of life on the earth [175, 176]. Over the past 100 years, the global temperature has increased by 0.8 °C and is expected to reach 1.1–5.4 °C by the end of next century. On the other hand, aberrant weather events like changes in rainfall patterns, frequent droughts and floods, increased intensity and frequency of heat and cold waves and outbreaks of insect-pests and diseases profoundly affect many biological systems and ultimately human beings [176].

Extreme weather events, which occur in every agricultural region of the world, cause severe crop damage. The persistent drought in the African Sahel has had the greatest human impact. In the USA, economic damage from single events can exceed $1 billion. The most severe, recent weather-related events were the drought of 1988 and the flood of 1993 [177].

Despite remarkable improvements in technology and crop yield potential, crop production remains highly dependent on climate because solar radiation, temperature and precipitation are the main drivers of crop growth [177]. Climate change, a long-term shift in the statistics of weather, has a significant impact on agricultural pests. The last decade of the twentieth century and the beginning of the twenty-first century have been the warmest period in the entire global temperature record [178]. The fourth assessment Report of the Intergovernmental Panel on Climate Change concluded that most of the observed increase in the average global temperature since the mid-twentieth century is very likely due to the observed increases in the anthropogenic greenhouse gas concentrations. It is now recognized that climate change will affect plant pests and diseases together with other components of change, i.e. anthropogenic processes such air, water and soil pollution, long distance introduction of exotic species and urbanization [179]. Climate changes affect plants in natural and agricultural systems throughout the world [180]. Climate change directly impacts crops as well as their interactions with microbials [181].

Climate has a significant effect on agricultural pests and determines the extent of economic losses caused by insects, pathogens and weeds. Globally, in 1995, annual soyabean production was valued at US$24.2 billion. Estimated losses caused by pathogens, insects and weeds were $3.2, $3.7 and $4.7 billion respectively as reported [182]. Climate change is expected to significantly increase those losses in many regions of the world.

It has been reported that global climate warming may lead to altitude-wise expansion of the geographic range of insect pests [183], increase in abundance tropical insect species [184] decrease in the relative proportion of the temperature sensitive insect population [185], increased incidence of insect-transmitted plant diseases through range expansion and rapid multiplication of insect vectors [186]. Therefore, with a changing climate it is expected that farmers growing crops will face new and intense pest problems as global warming intensifies in years to come [187, 188].

The spatial and temporal distribution and proliferation of insects, weeds and pathogens is determined to a large extent, by climate, because temperature, light and water are major factors controlling their growth and development. The geographical ranges of several important insects, plant pathogens and weeds in the USA have recently expanded, including the SCN (*H. glycines*) and soybean sudden death syndrome (*Fusarium solani* f. sp. *glycines*) [181, 189, 190]. For example, the SCN range expanded from the southern US states in 1979 to the northernmost states bordering Canada in 1988. The soybean sudden death syndrome expanded from small, isolated areas in eastern Arkansas, and near the East Coast of North Carolina, to covering most of the eastern states [177].

The major drivers of climate change are (1) increased temperatures, (2) elevated CO_2 levels, (3) precipitation changes and (4) hurricanes, typhoons and wind currents. We will now discuss these four drivers.

Increased temperatures

The warming trend and changes in extremes can be expected to affect the regional incidence of weeds, insects and crop pathogens. A change in the patterns of precipitation for crop–pest interactions may be even more important than a change in the annual total rainfall [177]. The water regime of pests is vulnerable to a rise in the daily rate of and seasonal pattern of evapotranspiration resulting from warmer temperatures, drier air or windier conditions. Projected temperature increases can be expected to induce earlier and faster development of crops, and cause increased pest damage at the sensitive earlier stages of crop development [177].

Warm winters increase the overwintering populations of all pests and insect vectors of plant diseases [181] pests. For example, warm winters increase the population of aphids (soybean aphid, *A. glycines* and other aphid species) that transmit soybean mosaic virus and increase the population and number of generations per year of the soyabean defoliator, the Mexican bean beetle, *Epilachna varivestis*, in the USA [177].

Insects are particularly sensitive to temperature because they are stenotherms (cold blooded). In general, insects respond to higher temperatures with increased development rates and with less time between generations. It has been estimated that with a 2 °C temperature increase, insects might experience one to five additional life cycles per season [191]. Therefore, global warming is expected to benefit many insect species, especially in temperate regions, where crop losses will increase.

Insect metabolism increases with temperature to a certain point. When metabolism increases insects have to eat more [192]. If the temperature is too cold or too hot, the insect population will grow more slowly. This is the reason why crop losses will be greatest in temperate regions, but less severe in the tropics. Temperate regions are not at the optimal temperature now, so if temperature increases there, insect populations will grow faster [192]. Therefore, soyabeans grown in temperate zones such as China, Japan and the USA will likely see increased insect damage with increased global warming, whereas soyabeans grown in the tropics, e.g. Africa and South America, will be less affected by global warming-induced insect damage.

In insect species, limited by low temperatures at higher latitudes, increasing temperatures reduce winterkill and result in a greater ability to overwinter, therefore extending the insect species' geographical range [183]. In Japan, warmer climates led to the northwards migration of the green stinkbug, *N. viridula*, a major agricultural pest damaging the fruiting structures of soyabean, rice, cotton and other crops [193].

As the globe warms, farmers will face more drought, flooding and crop-damaging heat. In addition, higher temperatures will produce more voracious defoliating insects e.g. grasshoppers, caterpillars and other crop-devouring pests, with potentially catastrophic consequences for the world's food supply. Scientists became keenly aware of the insect threat 10 years ago. That's when Curtis Deutsch et al. [194] published a study showing that as temperatures rise, nearly all insects will multiply and increase their metabolic rates.

To determine what kind of damage this increased insect appetite might have on the global food system, Deutsch and his team built a computer program that combined physiological data on hundreds of insect species with climate models. When the planet warmed by an average of 2 °C, as models predict will happen by 2100, if not sooner, wheat crops shrunk by 46%, rice by 19% and maize (or corn) by 31%. Temperate, productive regions like the United States' 'corn belt', wheat fields in France, and rice paddies in China were especially hard hit, the team reports in Science [195].

Increased temperatures, humidity and precipitation result in the spread of plant diseases as wet vegetation promotes the germination of spores and the proliferation of fungi and [188]. Under the warmer-but-drier conditions, projected for North America, crop losses due to plant diseases are expected to decline up to 30%. However, under wetter conditions projected for Africa, losses to diseases are expected to increase by more than 100% for certain crops.

Temperature is the most important factor influencing the biology of nematodes. Nematode developmental rate is directly influenced by temperature with slower development at cooler and faster growth at warmer soil temperatures. Therefore, an increase in atmospheric temperature, due to global warming, is expected to result in more generations per season and an expansion of their geographical range [188].

Elevated CO_2 levels

CO_2 concentration in the atmosphere has increased drastically from 280 to 370 ppm and is likely to be doubled by 2100 [176]. This change is attributed mainly to the overexploitation and misuse of natural resources for various anthropogenic developmental activities such as increased urbanization, deforestation and industrialization.

Global climate models have projected the effects of increasing atmospheric concentrations of greenhouse gases. Simulations predict mean global warming of between 1.5 and 5.8 °C (2.7 and 10.4 °F) by the end of the century. Because a warmer atmosphere can hold more water vapour a mean global increase in global precipitation of 5–15% is predicted. Also, a warming trend can be expected to affect the biophysical processes of photosynthesis and respiration, the regional infestations of weeds, insects and plant diseases and the thermal and hydrological regimes governing our agricultural systems [177].

Increased CO_2 levels can impact both the host plant and the pathogen and insect herbivore in a multiple of ways [178]. New pathogen races and insect biotypes may evolve rapidly under elevated temperature and CO_2, as evolutionary forces act on massive pest and pathogen populations under a favourable microclimate within an enlarged plant canopy [196]. Higher growth rates of leaves and stems observed for plants grown under high CO_2 concentrations result in denser canopies with higher humidity that favours pathogens. Denser canopies, which result in a greater biomass production and favourable microclimates, become more conducive for foliar pathogen development [188].

Pathogen growth can be directly affected by higher CO_2 concentrations resulting in increased fungal spore production. A high concentration of carbohydrates in the host tissue, due to elevated CO_2 levels, promotes the development of biotrophic fungi such as rust [197]. In general, increased plant density will tend to increase leaf surface wetness duration and regulate temperatures, therefore making infection by foliar pathogens more likely [198]. Elevated CO_2 levels can increase the extent of damage by leaf feeding insects, on soyabeans. During the early season soyabeans grown in elevated CO_2 atmosphere had 57% more damage from insects (including the Japanese bean beetle, *Popillia japonica*; and the Mexican bean beetle, *E. varivestis*) than soyabeans grown in an ambient atmosphere. The increased feeding under elevated CO_2 levels required that an insecticide be applied to be able to complete the experiment. The researchers surmised that measured increases in the levels of simple sugars in the soyabean leaves may have stimulated the additional insect feeding [199]. Increased atmospheric CO_2 is expected to alter the nutritional composition of crops, thereby affecting the severity insect attack.

Elevated levels of CO_2, equivalent to those projected to occur under global climate change scenarios, increase the susceptibility of soyabean foliage to herbivores by down-regulating the expression of genes related to the defense hormones, jasmonic acid and ethylene; these in turn decrease the gene expression and activity of cysteine proteinase inhibitors (CystPIs), the principal antiherbivore defenses in foliage [200].

To determine the effects of elevated CO_2 on the preference of the Japanese beetle (*P. japonica*) for soyabean leaves of different ages within the plant, a study was conducted at the University of Illinois [200]. When given a choice, the Japanese beetle inflicted greater levels of damage on older leaves than on younger leaves, and there was a trend for a greater preference for young leaves grown under elevated CO_2, compared with those grown under ambient CO_2. The more damaged older leaves and those grown under elevated CO_2 had reduced CystPI activity. CystPIs are potent defenses against the Japanese beetle, and the effectiveness of this defense is modulated by growth under elevated CO_2, as well as leaf position [199, 201, 202].

Enhanced CO_2 levels increase the biological control activity of generalist predators [203]. The herbivorous cotton bollworm (*H. armigera*) larvae feeding on elevated CO_2-grown pea (*Pisum sativum*) at 700 ppm were significantly smaller than those grown on ambient CO_2-grown plants. The omnivorous, spined predatory shield bug (*Oechalia schellenbergii*) performed best when fed on bollworm larvae from the elevated-CO_2 treatment, apparently because the *H. armigera* prey was smaller and thus easier for the predator to overcome. Taken together, results indicate that elevated CO_2 may benefit generalist predators through increased prey vulnerability, which would put pest species under higher risk of predation [203].

Global warming means more pests, including weeds [204]. Plants are divided into C3 and C4 types, according to how they utilize CO_2, in photosynthesis. Wheat, rice and soyabeans are C3 plants. C3 plants respond to increased CO_2 concentration with increased growth. Maize, sorghum and sugarcane are C4 plants and are less responsive to an increase in CO_2. Weeds also follow this division [204]. Therefore, increased CO_2 levels encourage the growth of C3 weeds and increase the water use efficiency of both C3 and C4 weeds. Increased temperatures promote the growth of C4 weeds [205]. For this reason, subtropical C4 weeds in the USA are likely to spread northwards as winter temperatures in northern climes become warmer with global warming. Also, it is predicted that, perennial weeds in soyabean and other crops may be more difficult to control, since increased photosynthesis, due to increased CO_2, may lead to greater storage of food supplies.

Precipitation changes

Precipitation – whether optimal, excessive or insufficient – is a key variable that affects crop–pest interactions [188]. Some insects are sensitive to precipitation and are killed or removed from crops by heavy rains. Drought stress has been reported to result in increased pest outbreaks. It has been shown that drought stress can affect the physiology of plant host species, leading to changes in the insects that feed on them [206]. Abnormally cool, wet conditions can also cause severe insect infestations.

Increased precipitation, due to heavy rains, benefits epidemics and prevalence of leaf fungal pathogens [188]. The heavy rains in 1993, which caused flooding and damaged crops in over 11 million acres, caused fungal epidemics in maize, soyabean, alfalfa and wheat in the US Great Plains. Heavy rains in north central USA in 1993, caused outbreaks of soybean sudden death syndrome, caused by the soil-borne root pathogen, *F. virguliforme*, formerly known as *F. solani* f. sp. *glycines* [188].

Fungal pathogens of insects are favoured by high humidity and their incidence is increased by climate changes that lengthen periods of high humidity and reduced by those that result in drier conditions [188]. Chen and McCarl [207] investigated the relationship between precipitation and pesticide costs in the USA and concluded that increases in rainfall lead to increases in average pesticide costs for cotton, maize, soyabean and wheat.

The severe drought of 1988 in the US Midwest, accompanied by higher than normal temperatures, began early in the spring and continued throughout most of the summer [208]. Crop yields dropped by approximately 37% and required a $3 billion government bailout for farmers. The drought impacted crop pests, with outbreaks of the two-spotted spider mite (*Tetranychus urticae*) damaging soyabeans throughout the entire Midwest region. Mite damage occurred during the critical flowering, pod development and pod filling growth stages. To control the mite, insecticide was applied to about 3.2 million hectares. Losses to Ohio farmers alone were estimated at $15–20 million [209].

Water stress, due to drought, decreases plant vigour and alters the C/N ratio which in turn lowers the level of plant resistance to the feeding of nematodes and insects. Outbreaks of the SCN (*H. glycines*) correlated with drought conditions in the north central USA in 1990 [177].

Hurricanes, typhoons and wind currents

Hurricanes and wind currents provide large-scale transportation for disease agents (e.g. fungal spores) and insects from overwintering areas [181]. Asian soybean rust (*P. pachyrhizi*) is an exceptionally aggressive pathogen that has rapidly spread globally since about 2000, moving from Asia through Africa and on to South America [210]. In September 2004, hurricane Ivan struck the US Gulf Coast introducing *P. pachyrhizi* into Louisiana, and by December 2004 there were 20 new detections in eight states, outlining the periphery of the hurricane impact zone [211].

Rise and Demise of Soyabean Pest Management: The Brazilian Example

Introduction

The history and evolution of the soyabean IPM programme in Brazil, like elsewhere in the world, is linked to the abusive use of pesticides and the subsequent results [212, 213]. The over-use of pesticides led to government policies to mitigate the side effects of pesticides, and a major component of this change was the implementation of IPM programmes [214].

The chronological development of the soyabean pest management programme in Brazil can be broken down into four eras: (1) the beginning years – 1970s, (2) the Baculovirus era – 1980s, (3) the 1990s – egg parasitoid era and (4) soyabean insect management in the new millennium. This section discusses the tremendous level of adoption of soyabean pest management by Brazilian producers (2 million hectares under soyabean pest management in the 1980s) [215] and the subsequent decrease in the current area under soyabean pest management (almost nil) due to changes in agronomic practices [214].

The beginning years – 1970s

Soyabean pest management was perhaps the most successful pest management programme developed in Brazil [214] and most likely the largest, most prominent and successful soyabean pest management programme in the world. However, it was initially an insect management programme and it was not until the later years that diseases and weeds were integrated into the programme resulting in an IPM programme.

The commercial cultivation of soyabeans in Brazil began expanding in the 1970s. The predominant method of pest control was exclusively based on the use of insecticides and acaricides [216, 217]. During this period an average of five pesticide applications (varying from three to ten) were made per soyabean crop [218]. The insecticides applied, such as dieldrin, DDT, toxaphene and methyl parathion were broad spectrum products and highly toxic to humans and the natural enemies [218].

In 1972, the Brazilian government, in collaboration with the University of Wisconsin/USAID, established the Projeto Nacional da Soja (National Soybean Project) with headquarters at the Rio Grande do Sul State Department of Agriculture in Porto Alegre. In 1973, with the establishment of EMBRAPA, the project became the EMBRAPA-Soja Program. In 1975, the program moved to Londrina and joined the Agronomic Institute of Paraná-IAPAR. There, the EMBRAPA-Soja program, EMATER-Parana, and some Brazilian and US universities began pioneering work on IPM in soyabean, primarily to the promote the rational use of insecticides to growers. The Porto Alegre project provided some of the basic information later used in the development of pest management components and provided opportunities for Brazilian students to conduct MS theses on soyabean pest management subjects. Of the numerous students who conducted research in the areas of entomology, nematology, plant pathology, weed science and plant breeding, many have had long careers at EMBRAPA, and have retired, and some are still active in soyabean research. These scientists are the ones that are responsible for the development of the Brazilian soyabean pest management programme.

Due to the overuse of pesticides in Brazilian soyabean production institutions such as EMATER-PR, the Agronomic Institute of Paraná-IAPAR, and some universities began pioneering work on IPM in soyabean [34]. IPM was based on the premise that cultivated plants can tolerate certain levels of injury without economically significant yield reductions; and insecticides should be used only as a complementary method, since natural biological control must be responsible for keeping pests under control. Thus, in the 1970s, economic thresholds were established for the major soyabean pests, the defoliating caterpillars and seed sucking stink bugs.

Pilot projects were conducted with extension agents to test the IPM technologies developed. Brazilian personnel were trained on the use of selective insecticides for the protection of natural enemies [34]. The first major impact was a reduction in the amount of insecticide used to control pests. The IPM concepts were then rapidly adopted by the extension personnel who passed the message on to the growers. After only 3–4 years of soyabean IPM adoption in Brazil, the application of pesticides in soyabean was reduced from an average of five applications to only two applications per season [214]. The soyabean IPM approach radically changed the common practice of expensive, preventative, calendar-based insecticide applications to a more economically profitable and environmentally safe insect control strategy [214].

The Baculovirus era – 1980s

In the 1970s and 1980s, the major lepidopterous soyabean pest in southern Brazil was the velvetbean caterpillar, *A. gemmatalis* [219, 220] which caused severe defoliation of soyabean plants [214]. The fungal entomopathogen, *M.* (*Nomuraea*) *rileyi* [221] is responsible, especially in rainy seasons, for maintaining *A. gemmatalis* populations under control and when the infestation is timely, virtually eliminates the requirement of insecticide to control these pests [39].

Another naturally occurring biocontrol agent, a nuclear polyhedrosis virus, *Baculovirus anticarsia*, was also found to be effective in providing natural control [222, 223]. This virus was referred to as AgNPV (*A. gemmatalis* nuclear polyhedrosis virus). A pilot programme to develop the use of AgNPV was developed and implemented [224]. The pilot project, initially tested in the states of Parana and Rio Grande do Sul, rapidly spread to other states and reached nearly 1.6 million hectares, representing about 10% of the total area cultivated with soyabean in Brazil [178]. The AgNPV initially applied consisted of a homemade solution made from field collected diseased *A. gemmatalis* larvae (50 larvae equivalents/ha). Later, a wettable powder formulation was developed and widely used [187]. In the early 2000s, an area of approximately 2.0 million hectares was treated with baculovirus, representing a savings of about 2 million litres of insecticides. The addition of the AgNPV component dramatically increased the effectiveness and success of the Brazilian soyabean IPM programme. Due to the success of the programme in Brazil, it was adopted by other Latin American countries [225].

The 1990s – egg parasitoid era

In the 1990s, a new control tactic, the biological control of soyabean stink bugs with egg parasitoids was added to the Brazilian soyabean IPM programme. The objective was to achieve a more stable control by augmenting the number of these beneficial agents and by conserving field populations of stink bug parasitoids [226].

Thirty-two species of stink bugs (Hemiptera: Pentatomidae) have been reported in soyabean in Brazil [50] and several are considered as important soyabean pests. They feed on the developing grains which results in foliar retention [227], reduced grain quality [228] due to the transmission of yeast spot disease, *N. coryli* [229], and severe yield reduction [230–232]. There are more than 20 species of parasitoids providing natural biological control of stink bugs in Brazilian soyabean fields [233]. The most important species are *Trissolcus basalis*, that preferentially attacks *N. viridula* eggs, and *Telenomus podisi* that prefers eggs of *E. heros* and *P. guildinii* [214]. However, these parasitoids tend to occur in high populations, when the stinkbugs have already reached economic threshold levels. For this reason, a programme for laboratory production and inoculative release was developed at the Embrapa Soyabean project at Londrina, PR [234].

After 4 years of field tests, a pilot programme was developed in collaboration with the Paraná state extension service, using *T. basalis* [235]. Cards with *T. basalis* parasitized stinkbug eggs were placed at the edge of soyabean fields (where stinkbug infestations generally begin), at the rate of 5000 egg parasitoids/ha, at the final flowering stage [236].

To increase the effectiveness of egg parasitoids for stink bug control, growers were recommended to use AgNPV to control the velvetbean caterpillar. To control other lepidopterous defoliators, and to avoid killing egg parasitoids that colonize the fields early in the season, growers were recommended to use microbial and/or selective insecticides such as those based on *Bacillus thuringiensis* or insect growth regulators (e.g. diflubenzuron).

Following the successful results of the pilot project, the stinkbug parasitoid technology was implemented at the farm level in the 1993/94 season. In 1994/95 a modified programme was deployed in the micro river basins of Paraná state consisting of 18 020 ha and 343 soyabean producers. Before implementation of the project, stinkbug parasitism was low [50]. After implementation of the IPM programme 90% of *N. viridula*, 65% of *P. guildinii* and 78% of *E. heros* eggs were killed by egg parasitoids [237]. The mean number of insecticide applications per cropping year in the river basin fell from 2.8 (1993/94 season) to 1.23 four seasons later. Area under biocontrol of the velvetbean caterpillar with AgNPV increased 57%, from 205 ha in the 1993/94 season to 2730 ha in 1998 [238].

It is important to note that during the 1990s, several other components were added to the soyabean IPM programme in Brazil. This included economic thresholds to control the coleopterous stem borer, *Sternechus subsignatus* [39] and the application of insecticides, only on the border rows, or at a reduced dosage, mixed with sodium chloride to control stink bugs [239]. After the adoption of soyabean IPM in the 1980s, the use of insecticides in Brazil was reduced to about two applications per season [240].

Soyabean Insect Management in the New Millennium

Over the last 40 years, soyabean yield in Brazil has doubled, increasing from an average of <1500 kg/ha in the 1970s to more than 3000 kg/ha in 2015 [240]. The yield increase, along with higher soyabean prices, the lower cost of many insecticides, the change in agronomic practices and the failure to use economic thresholds and pest monitoring [241], have led to the abandonment of soyabean IPM in Brazil. Consequently, insecticide applications again reached an average of four to six and even more sprays per crop cycle, impairing the effectiveness of all existing biological control agents for soyabeans [242].

What specifically were the factors that led to the demise of soyabean IPM in Brazil? Why wasn't the IPM programme sustainable? In the 1970s, at the beginning of the soyabean IPM implementation in Brazil, the conventional cultivation system was practiced on 100% of the soyabean area. This cultivation system included several machinery operations to till and level the soil. This system was eventually abandoned, mainly due to the severe erosion, loss of soil nutrients, and due to economic reasons. Then, the no-tillage cultivation, a new system was proposed and implemented. Under this system the soil is left undisturbed and covered with crop residues from the previous wheat crop. The change was dramatic: from <5000 ha under no-tillage at the beginning of the 1970s to 32 million ha in 2013. This change, although Q9 important from an ecological and economical point of view, severely impacted the IPM system practiced by soyabean growers [214].

Because of no-till cultivation, many soil-inhabiting pests increased in importance and were damaging soyabean plants at the beginning of the cultivation season. To control these pests, insecticide applications were required and these applications disrupted the needed balance of pests versus natural enemies. As a result, polyphagous pests that live on or in the ground such as the root sucking burrower, *Scaptocoris castanea* [243], tropical brown stink bug, *E.* heros, [244], green belly stink bugs, *Dichelops furcatus* and *D. melacanthus* and several species of root-feeding beetles [245] were favoured and their populations significantly increased. This was essentially the beginning of the end of soyabean IPM in Brazil.

The current system of multiple cropping has also contributed to increasing pest populations and was another dagger in the heart of an already weakened soyabean IPM programme. Multiple cropping with soyabean and/or maize double cropping without a break, provides a food resource for pests throughout the year. These pests needed to be controlled, if high yields were to be achieved, and the major control agent was pesticides that destroyed the natural enemy population.

The next dagger, in the heart of the IPM programme, was the unsustainability of the use of the biological control agent, AgNPV, to control the velvetbean caterpillar. Because of the difficulty of producing AgNPV the demand was much greater than the supply [15]. Lack of information about the use of AgNPV was the primary cause for the unsuccessful application of this biological control agent. First, the growers who were accustomed to the fast action of insecticides did not appreciate the delayed mode of action of AgNPV. Second, most growers, many with extremely large fields, were unprepared for the careful monitoring of A. *gemmatalis* phenology necessary to ensure effective application of AgNPV [225]. For these reasons, in regions where the extension programme was weak, the use of the virus was negligible [246].

Another setback for soyabean IPM in Brazil was a reduction in the costs of conventional insecticides which were toxic to natural enemies of soyabean pests, and their subsequent increased use. In addition, the mixture insecticides with herbicides to control weeds exacerbated the problem. Moreover, because of the increased insecticide use, secondary pests such as chewing caterpillars that feed on pods and leaves (*Spodoptera* spp. and soybean looper, *Pseudoplusia includens*), and mites, thrips and whiteflies increased in their abundance and gained the status of major pests [214].

Finally, the arrival and spread of the soybean rust fungus, *P. pachyrhizi*, had a significant impact on the soyabean IPM programme. Some of the most effective fungicides for controlling rust were harmful to the naturally occurring entomopathogenic fungi such as *M.* (*Nomuraea*) *rileyi* and others [247]. This added even more pressure to the already weak role that the natural enemies and insect pathogens were already playing due to the change in cultural practices. The various factors mentioned above led to a complete collapse in the use of the classical and highly successful soyabean IPM programme in Brazil by the end of 2010 [214].

Recently, consideration of IPM has been revived with the introduction of a new pest in the Americas. In Brazil, *H. armigera* was identified for the first time in February 2013 [248]. Damage estimates reached US$0.8 billion in the first crop season. The Brazilian government, throughout its institutions, established a task force to train most of the growers and consultants to identify and control the pest correctly.

An encouraging factor is that the private industry is providing strong support in the promotion of soyabean IPM in Brazil. For example, during the last season (2016/2017) the egg parasitoid, *Trichogramma pretiosum*, was released in more than 2 million ha of soyabean in Brazil for the biological control of *H. armigera*. In fact, the demand is so high that the companies producing the *Trichogramma* are not able to meet the farmers' requests. In addition, this crop season (2017/18), the company, BUG – Piracicaba, will sell the stink bug egg parasitoid *T. podisi*. Again, the production will not be able to meet the farmer demand because of the difficulty of production. Rearing is difficult because the egg host needs to be stored in liquid nitrogen (F. Goncalves, Personal Communication).

Therefore, if on the one hand, *H. armigera* brought some yield losses to Brazilian growers in its first years in the country, on the other hand, the presence of this pest brought back the need to employ some old IPM concepts that had almost been forgotten by most growers. With the presence of *H. armigera* in Brazil, growers are learning that to address the insect problems and still maximize agricultural production.

With the development of transgenic soyabeans for insect and weed control, the presence of invasive pests such as *H. armigera*, the planting of early maturity varieties and indeterminate cultivars, and the availability of environmentally friendly biopesticides and egg parasitoids, agronomic practices (e.g. crop rotation and the new conventionally bred pest resistant varieties), and reliable economic thresholds [241, 249, 250], there is an urgent need to design modern, sustainable soyabean IPM programmes for Brazilian growers [214].

This subject has been discussed in several forums [214]. There is a consensus, among Brazilian scientists, that there is an urgent need that the government develop legislation that promotes IPM as an official governmental policy and to stimulate governmental funding agencies to support IPM, not only for soyabeans, but also for all major commodities. It is envisaged that that private industries should collaborate with publicly supported agencies, international agencies and NGOs, to support on a regional scale, IPM programmes that yield long-term benefits, not only to growers, but also to the broader community [214].

However, Brazilian IPM scientists have a monumental barrier to overcome in implementing a return to IPM in Brazil. Governmental policies have significantly eroded what remained of IPM in Brazil. In 2013, the last year figures are available, Brazilian buyers purchased $10 billion worth, or 20% of the global pesticide market. Since 2008, Brazilian pesticide demand has risen 11% annually – more than twice the global rate [251].

The Brazilian farmers have become the world's top exporters of sugar, orange juice, coffee, beef, poultry and soyabeans. This rapid growth has made Brazil an enticing market for pesticides, banned or phased out in richer nations, because of health or environmental risks. In addition, they have earned a more dubious distinction: in 2012, Brazil passed the United States as the largest buyer of pesticides [251].

This excessive, and misuse, of pesticides has led to many cases of insecticide poisoning and contamination of the food supply. One factor blocking safeguards, that are more forceful, is Brazil's increasingly powerful agricultural lobby. The industry's influence, and tight budgets for regulators, limits Brazil's ability to enforce pesticide regulations [251].

Consider the time it takes Anvisa, the federal agency in charge of evaluating pesticide health risks, to evaluate a proposal by a manufacturer to sell a pesticide in Brazil. By law, the agency is supposed to analyse a new chemical in no more than 120 days. However, Anvisa can take years to complete this process. With fewer than 50 scientists, compared with hundreds at comparable agencies in the United States or Europe, it has a backlog of more than 1000 chemicals awaiting review [251]. This severely limits the introduction of alternatives to pesticides, such as biological control agents and microbial agents, that are environmentally acceptable, in IPM programmes.

IPM programmes are greatly influenced by regulatory mechanisms in place to manage the handling and use of pesticides. These regulatory activities include pesticide registration, banning of unsafe pesticides, food safety, farm worker safety and prevention of pest resistance. Indeed, favourable governmental policies are the key to IPM adoption. If policies create barriers to IPM adoption, such that there is little economic incentive to adopt, there may be little return to IPM research and extension [252]. The Brazilian IPM scientists have their marching orders. Policies and actions affecting IPM adoption in Brazil need to be modernized in favour of consumers, growers and biodiversity.

Acknowledgement

Preparation of this review was supported by the USAID Cooperative Agreement No. AID-OAA-L-15-00001 awarded to Virginia Tech.

Q10 References

1. Willis H. Growing Great Soybeans. *Acres USA* March 1989; pp. 1, 6–8. Available from: URL: http://eap.mcgill.ca/CPSO_3.htm.
2. Hartman GL, West ED, Herman TK. Crops that feed the world 2. Soybean – worldwide production, use and constraints caused by pathogens and pests. Food Security 2011;3:5–17.
3. USDA/FAS. World Agricultural Production. Circular Series, WAP 10–17, Oct. 2017. Available from: URL: https://apps.fas.usda.gov/psdonline/circulars/production.pdf.
4. Wikipedia: Soybean. Available from: URL: https://en.wikipedia.org/wiki/Soybean 2017 (last accessed 11 November 2017).
5. Hammond RB, Higgins RA, Mack TP, Pedigo LP, Bechinski EJ. Soybean pest management. In: Pimentel D, editor. CRC Handbook of Pest Management in Agriculture. 2nd ed. CRC Press, Inc., Boca Raton, FL; 1991. p. 341–472.
6. Hammond RB. Soybean insect IPM. Radcliffe's IPM world textbook. Available from: URL: https://ipmworld.umn.edu/hammond (last accessed 11 November 2017).
7. Hartman GL, Sinclair JB, Rupe JC (editors). Compendium of Soybean Diseases. 4th ed. American Phytopathological Society, St. Paul; 1999. 201 p.
8. Hartman GL, Hill CB. Diseases of soybean and their management. In: Singh G, editor. The Soybean. CABI, 2010. p. 276–99.
9. Oerke EC, Dehne H-W. Safeguarding production-losses in major crops and the role of crop protection. Crop Protection 2004;23:275–85.
10. Oerke EC. Centenary review: crop losses to pests. Journal of Agricultural Science 2006;144:31–43.
11. Gupta RK, Chandel YS, Mehta PK. Loss in yield due to insect–pest complex in soybean. Journal of Insect Science (Online) 1997;10:72–3.
12. Wrather JA, Anderson TR, Arsyad DM, Tan Y, Ploper LD, Porta-Puglia A, *et al.* Soybean disease loss estimates for the top ten soybean-producing countries in 1998. The Canadian Journal of Plant Pathology 2001;23:115–21.
13. Queensland Department of Agriculture and Fisheries. Insect pest management in soybeans. Available from: URL: https://www.daf.qld.gov.au/plants/field-crops-and-pastures/broadacre-field-crops/integrated-pest-management/ipm-information-by-crop/insect-pest-management-in-soybeans. Last updated March 2011.
14. Hoffmann-Campo CB, Spalding Corrêa-Ferreira B, Moscardi F. Soja-manejo integrado de insetos e outros Artropods-praga. Brasilia: Embrapa; 2012. 859 pp. Available from: URL: http://www.cnpso.embrapa.br/artropodes/Capitulo0.pdf
15. Hoffmann-Campo CB, Oliveira LJ, Moscardi F, Corrêa-Ferreira BS, Lorini IA, Borges M, *et al.* Integrated pest management in Brazil. In: Maredia KM, Dakuo D, Mota-Sanchez D editors. Integrated Pest Management in the Global Arena. CABI Publishing, Cambridge, MA 02138; 2003. pp. 285–99.
16. Ontario. Insects and pests of field crops: soybean insects and pests. Agriculture, Food and Rural Affairs. OMAFRA Publication 811: Agronomy guide for field crops.
17. Formentini AC, Sosa-Gomez DR, de Paula Moraes SV, de Barrros NM, Specht A. Lepidoptera (Insecta) associated with soybean in Argentina, Brazil, Chile and Uruguay. Ciencia Rural 2015;45(12): Print version. Q11
18. Montgomery M. Grape colaspis: Some background. The Pest Management and Crop Development Bulletin; 2003. Available from: URL: http://bulletin.ipm.illinois.edu/pastpest/articles/200311b.html
19. Iowa State University. Soybean insects guide. Department of Entomology 2011. Available from: URL: https://www.ent.iastate.edu/soybeaninsects/colaspis_beetles (last accessed 11 November 2017).
20. Bessin R, Townsend L. Wireworms. ENTFACT-120: Wireworms. University of Kentucky, Department of Entomology. 2003. Available from: URL: https://entomology.ca.uky.edu/ef120. Uploaded 11/03.
21. Purdue University. Wireworms: *Melanotus* spp., *Agriotes mancus* Say, and *Limonius dubitans* LeConte. Purdue University Extension/Department of Entomology 2009. Available from: URL: https://extension.entm.purdue.edu/fieldcropsipm/insects/soybean-wireworms.php
22. Koch R, Wold-Burkness S. Soybean production: seed corn maggot in Minnesota soybean. University of Minnesota Agricultural Extension Service. 2015. Available from: URL: http://www.extension.umn.edu/agriculture/soybean/pest/seedcorn-maggot/
23. Purdue. Common soybean insects. Purdue University Cooperative Extension Service 1999. Available from: URL: https://extension.entm.purdue.edu/publications/E-85.pdf. Revised 11/99.
24. Oliveira LJ, Malaguido AB, Nunes J, Jr Corso IC, DeAngelis S, Farias LC, *et al.* Percevejos castanhos da raiz em sistemas de produção de soja. Embrapa-soja. Londrina 2000. Available from: URL: https://ainfo.cnptia.embrapa.br/digital/bitstream/CNPSO/2631/1/circtec28.pdf
25. Charleston K. Soybean stem fly outbreak in soybean crops. The Beatsheet-Insect pest management for Australia's Northern Region; 2013. Available from: URL: http://thebeatsheet.com.au/soybean-stem-fly-outbreak-in-soybean-crops/ (last accessed 26 May 2017).
26. Higley LG, Boethel DJ. (editors).Handbook of Soybean Insect Pests, Entomological Society of America; 1994. Q12

27. KudzuBug. Available from: URL: https://www.kudzubug.org/grower/ (last accessed 12 December 2017).

28. Wikipedia. Elasmopalpus. Available from: URL: https://en.wikipedia.org/wiki/Elasmopalpus

29. Chapin JW. Lesser cornstalk borer on soybean. Clemson Cooperative Extension; 2017. Available from: URL: https://www.clemson.edu/extension/publications/entomology/agricultural-importance/lesser-cornstalk-borer-on-soybean-ag26.html (last accessed 26 May 2017). 2008; 64 p.

30. Ohnesorg WJ, Hunt TE. Managing soybean defoliators. NebGuide G2259. University of Nebraska-Lincoln Extension, Institute of Agriculture and Natural Resources, May 2015.

31. Pinto AS, Parra JRP, Oliveira HN. Guia de campo de pragas e inseto beneficios da soja. ESALQ/USP: Piricicaba, Brasil (Ed). 2008. 64 p.

32. Mujica N, Alvites D, Carhuapoma P, Kroschel J. 4.3.2 Vegetable leafminer, *Liriomyza sativae* (Blanchard 1938). Pest distribution and risk atlas for Africa 2017. Available from: URL: https://cipotato.org/riskatlasforafrica/liriomyza-sativae/

33. Featured creatures: Vegetable leafminer. University of Florida. Available from: URL: http://entnemdept.ufl.edu/creatures/veg/leaf/vegetable_leafminer.htm

34. Bartolotto OC, Pomari-Fernandes A, de Bueno COFR, de Bueno AF, da Kruz Ynaira KS, Queiroz AP, *et al.* The use of soybean integrated pest management in Brazil: a review. Agronomy Science and Biotechnology 2015;1(1):25–32.

35. Ragsdale DW, Voegtlin D, O'Neill R. Soybean aphid biology in North America, Annals of the Entomological Society of America 2004;97:204–8.

36. Hill CB, Li Y, Hartman GL. Resistance of *Glycine* species and various cultivated legumes to the soybean aphid (Homoptera: Aphididae). Journal of Economic Entomology 2004;97:1071–77.

37. Domier LL, Latorre IJ, Steinlage TAM, Coppin N, Hartman GL. Variability and transmission of *Aphis glycines* of North American and Asian soybean mosaic virus isolates. Archives of Virology 2003;148:1925–41.

38. Kim KS, Hill CB, Hartman GL, Miam MAR, Diers BW. Discovery of soybean aphid biotypes. Crop Science 2008;48:923–8.

39. Hofmann-Campo CB, Oliveira LJ, Moscardi F, Gazzoni DL, Corrêa-Ferreira BS, Lorini IA, *et al.* Integrated pest management in Brazil. In: Maredia KM, Dakouo D, Mota-Sanchez, editors. Integrated Pest Management in the Global Arena. CABI Publishing, Wallingford and Cambridge;
Q13 2003. p. 285–99.

40. Gazzoni DL, Sosa Gomez DR, Moscardi F, Hoffmann-Campo CB, Correa-Ferreira BS. Insects, In: Tropical soybean: improvement and production; FAO Plant Production and Protection Series (FAO), no. 27/Empresa Brasileira de Pesquisa Agropecuaria, Londrina (Brazil). Centro Nacional de Pesquisa de Soja; 1994; p. 81–108.

41. Barbara KA. Featured creatures: Velvetbean caterpillar. Publication no. EENY-151, University of Florida/IFAS 2014; Available from: URL: http://entnemdept.ufl.edu/creatures/field/velvetbean.htm

42. Kogan M. Integrated pest management: historical perspectives and contemporary developments. Annual Review of Entomology 1998;43:243–70.

43. Watson JR. Life history of the velvetbean caterpillar (*Anticarsia gemmatalis* Hübner). Journal of Economic Entomology 1916;9:521–8.

44. Roberts P, Guillebeau P. Velvetbean caterpillar – *Anticarsia gemmatalis* (Hübner). Georgia IPM – integrated pest management 1999.

45. Purdue University. Field crops IPM: Soybean thrips. College of Agriculture/Entomology/Extension 2009.

46. Sharma HC. Bionomics, host plant resistance, and management of the legume pod borer, *Maruca vitrata* – a review. Crop Protection 1998;17:373–86.

47. Sharma HC, Saxena KB, Bhagwat VR. The legume pod borer, *Maruca vitrata*: bionomics and management. Information Bulletin no. 55. Patancheru 502 324, Andhra Pradesh, India: International Crops Research Institute for the Semi-Arid Tropics 1999. 42 p.

48. Srinivasan R. Insect and mite pests on vegetable legumes: a field guide for identification and management. AVRDC – The World Vegetable Center, Shanhua, Taiwan 2014; AVRDC Publication No. 14-778. 92 pp. Available from: URL: https://avrdc.org/download/publications/manuals/eb0228.pdf

49. Admin. *Etiella* spp., soybean pod borer pest and its control method. Indonesian Center for Food Crops Research and Development/Indonesian Agency for Agriculture Research and Development 2014. Available from: URL: http://pangan.litbang.pertanian.go.id/en/berita-497-etiella-spp-hama-penggerek-polong-kedelai-dan-cara-pengendaliannya.html (last accessed 10 June 2017).

50. Panizzi AR, Slansky F. Jr Review of phytophagous pentatomids (Hemiptera: Pentatomidae) associated with soybean in the Americas. Florida Entomologist 1985;68(1):184–214.

51. Corrêa-Ferreira BS, Moscardi F. Biological control of stink bugs by inoculative releases of *Trissolcus basalis*. Entomologia Experimentalis et Applicata 1996;79:1–7.

52. Muniappan R, Shepard BM, Carner GR, Ooi PAC. Arthropod Pests of Horticultural Crops in Tropical Asia. CABI, Wallingford, Oxfordshire; 2012.

53. Panizzi AR. Stink bugs on soybean in Northeastern Brazil and a new record on the southern green stink bug, *Nezara viridula* (L.) (Heteroptera: Pentatomidae). Neotropical Entomology 2002;31:331–2.

54. Corso IC, Heinrichs EA, Lehman PS. The occurrence of the fungus *Nematospora coryli* Peglion in bugs attacking soybean. Anais da Sociedade Entomologica do Brasil 1975;4(1):49–52.

55. Samaniotto LF, Panizzi AR. Interactions of selected species of stink bugs (Hemiptera: Heteroptera: Pentatomidae) from leguminous crops with plants in the neotropics. Florida Entomologist 2015;98:7–17.

56. Turnipseed SG, Kogan M. Soybean entomology. Annual Review of Entomology 1976;21:247–82.

57. Temple Joshua H. Redbanded stink bug, Piezodorus guildinii (Westwood): pest status, control strategies, and management in Louisiana soybean. [LSU doctoral dissertations]. 3691. 2011. Available from: URL: http://digitalcommons.lsu.edu/
gradschool_dissertations/3691. Q14

58. Panizzi AR, Slansky Jr F. New host plant records for the stink bug *Piezodorus guildinii* in Florida (Hemiptera: Pentatomidae). Florida Entomologist 1985;68:215–16.

59. Panizzi AR. Performance of *Piezodorus guildinii* on four species of *Indigofera* legumes. Entomologia Experimentalis et Applicata 1992;63:221–8.

60. Vicentini R, Jimenez HA. El vaneo de los frutos en soja. INTA Serie Tecnicia, 47. 1977.

61. Galileo MH, Heinrichs EA. Avaliação dos danos causados aos legumes de soja *Glycine max* (L.) Merrill por *Piezodorus guildinii* (Westwood, 1837) (Hemiptera: Pentatomidae), em diferentes níveis e épocas de infestaçao. Anais da Sociedade
Q15 Entomológica do Brasil 1978;7:33–9.

62. Galileo MH, Heinrichs EA. Avaliação dos danos causados as sementes de soja *Glycine max* (L.) Merrill por *Piezodorus guildinii* (Westwood, 1837) (Hemiptera, Pentatomidae) em diferentes níveis e épocas de infestaçao. Anais da Sociedade Entomológica do Brasil 1978;7:40–6.

63. Panizzi AR, Smith JG, Pereira LAG, Yamashita J. Efeito dos danos de *Piezodorus guildinii* (Westwood, 1837) no rendimento e qualidade da soja. Anais Seminário Nacional Pesquisa de Soja 1979;2:59–78.

64. Heinrichs EA. Stink bug complex in soybean. In: Goodman RM, editor. Expanding the use of Soybean. Proceedings of a Conference for Asia and Oceania. INTSOY Series No. 10. College of Agriculture, University of Illinois, Urbana; 1976. p. 173–7.

65. Galileo MH, Heinrichs EA. Retenção foliar en plantas de soja, *Glycine max* (L.) Merrill, resultant de açao de *Piezodorus guildinii* (Westwood 1837) (Hemiptera, Pentatomidae) en differentes niveis e épocas de infestaçao. Anais da Sociedade Entomológica do Brasil 1978;7:75–88.

66. Pennsylvania State University. Brown Marmorated Stink Bug. Available from: URL: http://ento.psu.edu/extension/factsheets/brown-marmorated-stink-bug. Revised Feb. 2015.

67. Faúndez EF, Rider D. The brown marmorated stink bug *Halyomorpha halys* (Stål, 1855) (Heteroptera: Pentatomidae) in Chile. Arquivos Entomolóxicos 2017;17:305–7.

68. Wikipedia. Brown marmorated stink bug. Available from: URL: https://en.wikipedia.org/wiki/Brown_marmorated_stink_bug.

69. Herbert A. Brown marmorated stink bug in soybean, summary to date Virginia Soybean Update/VA Cooperative Extension. Available from: URL: https://blogs.ext.vt.edu/soybean-update/2011/09/09/brown-marmorated-stink-bug-in-soybean-summary-to-date-dr-ames-herbert/. Posted on September 9, 2011 by David Holshouser (last accessed 10 June 2017).

70. Sinclair JB, Shurtleff MC. Compendium of Soybean Diseases. The American Phytopathological Society Inc., St. Paul; 1975.

71. Hartman G, Murithi HM. Field Guide to African Diseases and Pests. Soybean Innovation Laboratory, University of Illinois, Urbana, IL; 2017.

72. University of Illinois. Integrated Pest Management. RPD No. 502-Bacterial foliage diseases of soybeans. U of Ill., Urbana-Champaign. 2002. Available from: URL: http://ipm.illinois.edu/diseases/series500/rpd502/index.html (last accessed 21 June 2017).

73. Plantwise 1. Plantwise Technical Factsheet: Bacterial blight of soybean (*Pseudomonas savastanoi* pv. *glycinea*). Plantwise Knowledge Bank. Available from: URL: http://www.plantwise.org/KnowledgeBank/Datasheet.aspx?dsid=44963 (last accessed 21 June 2017).

74. Myung I-S, Kim J-W, An SH, Lee JH, Kim SK, Lee Y-K, *et al.* Wildfire of Soybean caused by *Pseudomonas syringae* pv. *tabaci*, a New Disease in Korea. The American Phytopathological Society 2009;93:1214.

75. Geisler LJ. Anthracnose. CROPWATCH. IANR, Univ. of Nebraska-Lincoln. 2017. Available from: URL: http://cropwatch.unl.edu/plantdisease/soybean/anthracnose (last accessed 22 June 2017).

76. CABI. Invasive Species Compendium. *Phakopsora meibomiae* (Soybean rust). CAB International 2015. Available from: URL: http://www.cabi.org/isc/datasheet/40018 (last accessed 23 June 2017).

77. CABI. Invasive Species Compendium. *Phakopsora pachyrhizi*. (Soybean rust). CAB International 2016. Available from: URL: http://www.cabi.org/isc/datasheet/40019 (last accessed 12 April 2015).

78. Rupe J, Sconyers L. Soybean Rust. The Plant Health Instructor. 2008. Available from: URL: http://www.apsnet.org/edcenter/intropp/lessons/fungi/Basidiomycetes/Pages/SoybeanRust.aspx (last accessed 22 June 2017).

79. Schnepf R. Asian Soybean Rust: Background and Issues (PDF|70 KB) (CRS Report for Congress RL32225). Congressional Research Service, 2005.

80. Hartman GL, Sinclair JB. *Dactuliochaeta*, a new genus for the fungus causing red leaf blotch of soybeans. Mycologia 1988;80:696–706.

81. Hartman GL, Datnoff LE, Levy C, Sinclair JB, Cole DL, Javaheri F. Red leaf blotch of soybeans. Plant Disease. 1987;71:113–8.

82. Stewart RB. An undescribed species of *Pyrenochaeta* on soybean. Mycologia 1957;49:115–7.

83. Hartman GL, Sinclair JB. Red leaf blotch (*Dactuliochaeta glycines*) of soybeans (*Glycine max*) and its relationship to yield. Plant Path 1996;45:332–43.

84. Hartman GL. *Phoma glycinicola* (Red leaf blotch): a threat to soybean production in the USA. In: Liu D, editor. Manual of Security Sensitive Microbes and Toxins. CRC Press, Boca Raton, Florida; 2014. p. 801–6. Available from: URL: https://www.ars.usda.gov/research/publications/publication/?seqNo115=288645 (last accessed 26 June 2017).

85. Braun E. Phytophthora root rot. American Phytopathological Society. 2017. Available from: URL: http://www.apsnet.org/publications/imageresources/Pages/IW000025a.aspx (last accessed 26 June 2017).

86. University of Minnesota Extension. Crop diseases: Soybean diseases. 2017. Available from: URL: https://www.extension.umn.edu/agriculture/crop-diseases/furtherinfo/ (last Accessed 27 June 2017).

87. Corn + Soybean Digest. The real threat to soybean yields: Phytophthora and Pythium. Available from: URL: http://www.cornandsoybeandigest.com/soybeans/real-threat-soybean-yields-phytophthora-and-pythium 2017 (last accessed 27 June 2017).

88. Koenning SR. Soybean viruses, Note 9. NCSU Plant Pathology Extension 2005. Available from: URL: https://soybeans.ces.ncsu.edu/depts/pp/notes/Soybean/soy009/soy009.htm

89. Kameya M. Virus Diseases of Soybean in Southeast Asian Countries. Food and Fertilizer Technology Center and for the Pacific and Asian Region (FFTC), Taipei, Taiwan. 2017. Available from: URL: http://www.fftc.agnet.org/library.php?func=view&style=type&id=20110714104359.

90. Dugje IY, Omoigui LO, Ekeleme F, Bandyopadhay R, Lava Kumar P, Kamara AY. Farmers 'Guide to Soybean Production in Northern Nigeria'. International Institute of Tropical Agriculture. 2009. Available from: URL: https://books.google.com/books?id=RXU6SNmxCyUC&pg=PR2&source=gbs_selected_pages&cad=2#v=onepage&q&f=false.

91. Cho EK, Chung BJ, Lee SH. Studies on identification and classification of 18 soybean mosaic virus disease in Korea II. Etiology of a necrotic disease of *Glycine max*. Plant Disease Reporter 1977;61:313–7.

92. Plantwise Knowledge Bank. Soybean mosaic virus. Available from: URL: http://www.plantwise.org/KnowledgeBank/Datasheet.aspx?dsid=48750 (last accessed 21 July 2017).

93. Soybean research and information initiative. Soybean viruses. North Central Soybean Research Program, 2014. Available from: URL: http://www.soybeanresearchinfo.com/diseases/soybeanviruses.html.

94. Vemana K, Jain RK. New experimental hosts of *Tobacco streak virus* and absence of true seed transmission in leguminous hosts. Indian Journal of Virology: An Official Organ of Indian Virological Society 2011;21(2):117–27. Published online 6 March 2011. doi: 10.1007/s13337-010-0021-0.

95. Kaiser WJ, Wyatt JD, Pesho GR. Natural hosts and vectors of tobacco streak virus in Eastern Washington. Phytopathology 1982;72:1508–12.

96. Costa AS, Carvahlo AM. Studies on Brazilian tobacco streak. Phytopathologische Zeitschrift 1961;42:113–38.
Q16

97. Almeida AMR, Bergamin Filho A, Amorim L. Disease progress of soybean bud blight and spatial pattern of diseased plants. Zeitschrift für Pflanzenkrankheiten und Pflanzenschutz 1994;101:386–92.

98. Laguna IG, Rodriquez Pardina PE, Truol GA, Nieves J. Diseases of viral aetiology on soyabean (*Glycine max*) in Argentina. Fitopatologia Brasileira 1988;13:192–8.

99. Fagbenle HH, Ford RE. Tobacco streak virus isolated from soybeans, *Glycine max*. Phytopathology 1970;60:814–20.

100. Fourie H, De Waele D, Mc Donald AH, Mienle C, Marais M, De Beer A. Nematode pests threatening soybean production in South Africa, with reference to *Meloidogyne.* South African Journal of Science 2015. http://dx.doi.org/10.17159/sajs.2015/20140212.
Q17

101. Padgett G. Nematode Management in Soybean. LSU Ag Center 2011. Available from: URL: http://www.lsuagcenter.com/topics/crops/soybeans/diseases/atlas/nematode-diseases/nematode-management-in-soybean.

102. CABI. *Heterodera glycines* (soybean cyst nematode). Invasive species compendium 2017. Available from: URL: http://www.cabi.org/isc/datasheet/27027.

103. Riggs RD, Niblack TL. Nematode pests of oilseed crops and grain legumes. In: Evans T Webster,(editors. Plant Parasitic nematodes in Temperate Agriculture. CAB International, Wallingford, UK; 1993.

104. Riggs RD, Wrather JA (editors). Biology and Management of the Soybean Cyst Nematode. American Phytopathological Press, St Paul, USA; 199.

105. Gao X, Jackson TA, Hartman GL. Interactions between the soybean cyst nematode and *Fusarium solani* f. sp. *glycines* based on greenhouse factorial experiments. Phytopathology 2006;96:1409–15.

106. Wrather JA, Koenning SR, Anderson TR. Effect of diseases on soybean yields in the United States and Ontario (1999–2002). Online. Plant Health Progress; doi: 10.1094/PHP-2003-0325-01-RV. 2003. Available from: URL: http://www.plantmanagementnetwork.org/pub/php/review/2003/soybean/.

107. Wrather JA, Anderson TR, Arsyad DM, Gai J, Ploper LD, Porta-Puglia A, *et al.* Soybean disease loss estimates for the top 10 soybean producing countries in 1994. Plant Disease 1997;81:107–10.

108. Rich JR, Kinloch RA, Dunn RA. Soybean Nematode Management. 2008; ENY 003. Univ. of Florida/IFAS Extension. Available from: URL: http://nematology.ifas.ufl.edu/assaylab/Documents/Soybean.pdf.

109. Ichinohe M. Studies on the morphology and ecology of the soy bean cyst nematode, *Heterodera glycines*, in Japan. Report of the Hokkaido National Agricultural Experimental Station, No. 48, 1955, 59–64.

110. Inagaki H. Soybean nematodes. Technical Bulletin, ASPAC Food & Fertilizer Technology Center, Taiwan 1977; No. 31, 17 p.

111. Sciumbato GL. Southern United States soybean disease loss estimate for 1990. Proceedings of the Southern Soybean Disease Workers 1991;32–8.

112. CABI. *Hoplolaimus columbus*. Invasive Species Compendium Commonwealth Agricultural Bureau International (CABI), Wallingford, UK 2018. Available from: URL: http://www.cabi.org/isc/datasheet/27519.

113. Ferris H. *Hoplolaimus columbus* 2013. Available from: URL: http://nemaplex.ucdavis.edu/Taxadata/G063s1.aspx.

114. Koenning SR. Management of Columbia Lance Nematode on Soybean. NCSU Plant Pathology Extension 2000. Available from: URL: https://www.ces.ncsu.edu/depts/pp/notes/Soybean/soy004/soy004.htm.

115. Reikert HF, Henshaw GE. Effect of soybean, cowpea and groundnut rotations on root-knot nematode build-up and infestation in dryland maize. The African Crop Science Journal 1998;6:377–83. Available from: URL: https://tspace.library.utoronto.ca/handle/1807/23707.

116. Fourie H, McDonald AH, Loots GC. Plant–parasitic nematodes in field crops in South Africa 6: Soybean. Nematology 2001;3:447–54. Available from: URL: http://dx.doi.org/10.1163/156854101753250773.

117. Bridge J, Starr JL. Plant nematodes of Agricultural Importance. Academic Press, Boston, MA; 2007. 152 p. Available from: URL: http://dx.doi.org/10.1201/b15142.

118. Sikora RA, Greco N, Silva JF. Nematode parasites of grain legumes. In: Luc M, Sikorra RA, Bridge J, editors. Plant Parasitic nematodes in Subtropical and Tropical Agriculture. CABI, Wallingford; 2005. p. 259–318.

119. Taylor AL, Sasser JN. Biology, Identification and Control of Root-Knot nematodes (*Meloidogyne* species). North Carolina State University, Raleigh, NC; 1978.

120. Kirkpatrick TL, May ML. Host suitability of soybean cultivars for *Meloidogyne incognita* and *M. arenaria*. Journal of Nematology 1989;21:666–70.

121. Kinloch RA. Response of soybean cultivars to nematicidal treatments of soil infested with *Meloidogyne incognita*. Journal of Nematology 1974;6:7–11.

122. CABI. *Pratylenchus thornei*. Invasive Species Compendium. 2017 Available from: URL: http://www.cabi.org/isc/datasheet/43903.

123. Castillo P, Vovlas N. 2007. Pratylenchus (Nematoda: Pratylenchidae): Diagnosis, Biology, Pathogenicity and Management. Brill Academic Publishers, Leiden, Netherlands; 2007. 555 p.

124. Chitambar J. Nematodes-*Pratylenchus thornei* Sher & Allen 1953. 6/2 2016. Available from: URL: http://blogs.cdfa.ca.gov/Section3162/?p=2031.

125. Wang KH. Featured Creatures: *Rotylenchulus reniformis* Linford & Oliveira (Nematoda: Tylenchida: Tylenchoidea: Hoplolaimidae: Rotylenchulinae). University of Florida/IFAS. 2007. Pub. No. EENY-210. Available from: URL: http://entomology.ifas.ufl.edu/creatures/nematode/r_reniformis.htm.

126. Ayala A, Ramirez CT. Host-range, distribution, and bibliography of the reniform nematode, *Rotylenchulus reniformis*, with special reference to Puerto Rico. Journal of Agriculture of University of Puerto Rico 1964;48:140–60.

127. Linford MB, Oliveira JM. *Rotylenchulus reniformis*, nov. gen. n. sp., a nematode parasite of roots. Proceedings of the Helminthological Society of Washington 1940;7:35–42.

128. Robinson AF, Inserra RN, Caswell-Chen EP, Vovlas N, Troccoli A. *Rotylenchulus* species: identification, distribution, host ranges, and crop plant resistance. Nematropica 1997;27:127–80.

129. Syngenta. Avicta complete beans. Nematode, insect and disease protection for soybean 2011. Available from: URL: http://www.syngenta-us.com/prodrender/imagehandler.ashx?ImID=6efe50d1-58e9-48f3-8b12-f2a1f5863d50&fTy=0&et=8.

130. Ball A, Bazaya BR. Economics and yield as influenced by different weed management practices in soybean. Proceedings of an international conference, Natural Resource Management Ecological Perspectives, SKUAST-Jammu, India, 18–20 February 2016.

131. Devi K Nandini Singh Kh Lenin Mangang CNJS Arangba Singh N Brajendra Athokpam H Singh Singh A Dorendro. Effect of weed control practices on weed dynamics, yield and economics of soybean [*Glycine max* (L.) Merrill]. Legume Research 2016;39;995–8. Online ISSN: 0976-0571.

132. Moore M. Top 10 worst weeds in corn and soybeans. Farm Industry News 2012. Available from: URL: http://www.farmindustrynews.com/crop-protection/top-10-worst-weeds-corn-and-soybeans.

133. Ewell JJ, O'Dowd DJ, Bergelson J, Daehler CC, D'Antonio CM, Gomez LD, *et al.* Deliberate introductions of species: research needs – benefits can be reaped, but risks are high. BioScience 1999;49:619–30. JSTOR1313438. doi: 10.2307/1313438.

134. Williams JD, Meffe GK. Nonindigenous Species. Status and Trends of the Nation's Biological Resources. Reston, Virginia: United States Department of the Interior, Geological Survey 1998;1.

135. Britton NL, Brown A. An illustrated flora of the northern United States, Canada and the British Possessions 1913;2:6.

136. Schonbeck M. Weed Profile: Pigweeds (*Amaranthus* spp.) eXtension, USDA/NIFA, Cooperative Extension 2014. Available from: URL: http://articles.extension.org/invasive_species.

137. Blackshaw RE, Brandt RN. Nitrogen fertilizer rate effects on weed competitiveness is species dependent. Weed Science 2008;56:743–47. Available from: URL: http://dx.doi.org/10.1614/WS-08-065.1. (last accessed 10 September 2012).

138. CABI. *Amaranthus tuberculatus* (Rough-fruited water-hemp). Invasive Species Compendium. Wallingford, UK. 2015; Available from: URL: http://www.cabi.org/isc/datasheet/112200.

139. CABI. *Amaranthus viridis* (slender amaranth). Invasive Species Compendium, Wallingford, UK. 2017. Available from: URL: http://www.cabi.org/isc/datasheet/4654.

140. Go Botany. *Amaranthus viridis*- slender amaranth. New England Wildflower Society. Available from: URL: https://gobotany.newenglandwild.org/species/amaranthus/viridis/.

141. Grubben GJH, Denton OA. Plant Resources of Tropical Africa 2. Vegetables. PROTA Foundation, Wageningen; Backhuys, Leiden; CTA, Wageningen, 2004.

142. Wikipedia. *Amaranthus palmeri*. Available from: URL: https://en.wikipedia.org/wiki/Amaranthus_palmeri (last accessed 29 September 2017).

143. Sprague C. Palmer Amaranth Management in Soybeans 2013. Available from: URL: http://bulletin.ipm.illinois.edu/wp-content/uploads/2014/03/PalmerAmaranth-2.pdf.

144. Bensch CN, Horak MJ, Peterson D. Interference of redroot pigweed (*Amaranthus retroflexus*), Palmer amaranth (*A. palmeri*), and common waterhemp (*A. rudis*) in soybean. Weed Science 2003;51:37–43.

145. TYWKIWDBI (Tai – Wiki – Widbee). Palmer's amaranth – a 'superweed.' 2014. Available from: URL: http://tywkiwdbi.blogspot.com/2014/09/palmers-amaranth-superweed.html (last accessed November 2017).

146. Horak MJ, Loughlin TM. Growth analysis of four Amaranthus species. Weed Science 2000;48:347–55.

147. Leonard A. Monsanto's bane: the evil pigweed. Salon. 27 August 2008. Available from: URL: http://www.salon.com/2008/08/27/monsantos_bane/.

148. Hensleigh P, Pokorny M. Palmer Amaranth (*Amaranthus palmeri* S. Watson). CABI Invasive Species Compendium. Tobacco streak virus 2017. Available from: URL: https://www.cabi.org/isc/datasheet/48107.

149. Holm L, Pancho JV, Herberger JP, Plucknett DL. A Geographic Atlas of World Weeds. Wiley-Interscience, John Wiley & Sons, New York; 1979.

150. Holm L, Doll J, Holm E, Pancho J, Herberger J. World Weeds: Natural Histories and Distribution. John Wiley & Sons, Inc., New York; 1997.

151. Pohl RW. The grasses of Iowa. Iowa State Journal of Science 1966;40:341–73.

152. Rominger JM. Taxonomy of Setaria (Gramineae) in North America. Illinois Biol. Monogr. 29.

153. Stapf O, Hubbard CK. *Setaria*. In: Flora of tropical Africa, Prain (Ed.), London. 1962;9:768–866.

154. Dekker J. The Foxtail (*Setaria*) Species-Group, Invited Review. Weed Biology Laboratory, Agronomy Department, Iowa State University, Ames, IA 50011, USA. Available from: URL: http://agron-www.agron.iastate.edu/~weeds/PDF_Library/Jax_Pubs/FoxWSciRev7.18.03.pdf.

155. Swearingen J, Bargeron C. Invasive Plant Atlas of the United States. University of Georgia Center for Invasive Species and Ecosystem Health 2016. Available from: URL: https://www.invasiveplantatlas.org/subject.html?sub=6394 (last accessed 21 August 2017).

156. CABI. *Setaria faberi* (giant foxtail). Invasive Species Compendium. Wallingford, UK. 2015. Available from: URL: http://www.cabi.org/isc/datasheet/49767 (last accessed 21 August 2017).

157. Harrison SK, Williams SC, Wax LM. Interference and control of giant foxtail (*Setaria faberi*) in Soybeans (*Glycine max*). Weed Science 1985;33:203–8. Available from: URL: https://www.jstor.org/stable/4044334? (last accessed 21 August 2017).

158. Plantwise 3. Yellow foxtail (*Setaria pumila*). Plantwise Technical Factsheet. Plantwise Knowledge Bank. Available from: URL: http://www.plantwise.org/KnowledgeBank/Datasheet.aspx?dsid=49773.

159. CABI. *Setaria viridis* (green foxtail). Invasive Species Compendium. Wallingford, UK. 2015.

160. Wikipedia. *Setaria viridis*. Available from: URL: https://en.wikipedia.org/wiki/Setaria_viridis.

161. Douglas BJ, Thomas AG,. Morrison IN, Maw MG. The biology of Canadian weeds. 70. *Setaria viridis* (L.) Beauv. Canadian Journal of Plant Science 1985;65:669–90.

162. Sharma N, Gupta GK, Verma RK, Sharma OP, Bhagat S, Amaresan N, *et al.* Integrated Pest Management for Soybean. Directorate of Plant Protection, Quarantine & Storage CGO Complex, NH IV, Faridabad-121001, India 2014; p. 41.

163. Dhaliwal GS, Arora R. Principles of insect pest management. National Agricultural Technology Information Centre, Ludhiana-141 001, India 1996; 374 p.

164. Dhaliwal GS, Heinrichs EA. Critical Issues in Pest Management. Commonwealth Publishers, New Delhi; 1998; 287 p.

165. Nwilene FE, Nwanze KF. Cultural control of insect pests in tropical crops. In: Dhaliwal GS, Heinrichs EA, editors. Critical Issues in Pest Management. Commonwealth Publishers, New Delhi; 1998. p. 26–66.

166. Painter RH. Insect Resistance in Crop Plants. The MacMillan Co., New York, USA; 1951.

167. Genetically modified soybeans! Available from: URL: https://worldofgenetics.weebly.com/genetically-modified-soybeans.html (last accessed 1 December 2017).

168. Van Driesche RG, Bellows Jr TS. Biological Control. Kluwer Academic Publishers, Boston/Dordrecht/London; 1996; 547 p.

169. DeBach P, Rosen D. Biological Control by Natural Enemies. Cambridge University Press, Cambridge, UK; 1991.

170. Altieri MA, Nicholls CI. Biological control in agroecosystems through management of entomophagous insects. In: Dhaliwal GS, Heinrichs EA, editors. Critical Issues in Pest Management. Commonwealth Publishers, New Delhi; 1998; p. 67–86.

171. Nakeeran S, Renukadevi P, Aiyanathan KEA. Exploring the potential of *Trichoderma* for the management of seed and soil-borne diseases of crops. In: Muniappan R, Heinrichs EA, editors. Integrated Pest Management of Tropical Vegetable Crops. Springer, Dordrecht; 2016. p. 77–130.

172. Singh RK. *Trichoderma*: a bio-control agent for management of soil borne diseases. Agropedia; 2010. Available from: URL: http://agropedia.iitk.ac.in/content/trichoderma-bio-control-agent-management-soil-born-diseases.

173. Beneduzi A, Ambrosini A, Passaglia LMP. Plant growth-promoting rhizobacteria (PGPR): their potential as antagonists and biocontrol agents. Journal of Genetics and Molecular Biology 2012;35Suppl. 4:1044–51. Published online 18 December 2012.

174. Schmutterer H. Side-effects of neem (*Azadirachta indica*) products on insect pathogens and natural enemies of spider mites and insects. Journal of Applied Entomology 1997;121(1–5):121–8.

175. Abrol Y, Gadgil S, Pant GB (editors). Climate Variability and Agriculture. Narosa Publishing House, New Delhi; 1996.

176. IPCC. Climate change -impacts, adaptation and vulnerability. In: Parry ML, Canziani OF, Palutikof JP, van der Linden PJ, Hanson, CE, editors. Cambridge University Press, Cambridge, UK; 2007. 976 p.

177. Rosenzweig C, Yang XB, Epstein PR, Chivian E. Climate change and extreme weather events. Global Change and Human Health, 2001; 2(2):90–104.

178. Ahanger R, Bhat HA, Bhat TA, Ganie SA, Lone AA, Wani IA, *et al.* Impact of climate change on plant diseases. International Journal of Modern Plant and Animal Sciences 2013;1(3):105–15.

179. Regniere J. Invasive species, climate change and forest health. In: Schlichter T, Montes L, editors. Forests in Development: A Vital Balance. Springer, Berlin; 2012. pp. 27–37.

180. Stern N. The Economics of Climate Change: The Stern Review. Cambridge University Press, Cambridge, UK, 2007.

181. Rosenzweig C, Iglesia A, Yang XB, Epstein PR, Chivian E. Implications of Climate Change for U.S. Agriculture: Extreme Weather Events, Plant Diseases, and Pests. Cambridge, Massachusetts: Center for Health and the Global Environment, Harvard Medical School, Cambridge, MA; 2000. 56 p.

182. Oerke EC, Dehne HW, Schonbeck F, Weber A. Crop Production and Crop Protection: Estimated Losses in Major Food and Cash Crops. Elsevier, Amsterdam and New York; 1995. 830 p.

183. Ephinstone J, Toth I. Erwinia chrysanthemi (Dikeya spp.) – The Facts. Potato Council, Oxford, UK; 2008.

184. Diffenbaugh NS. Global warming presents new challenges for maize pest management. Environmental Research Letter 2008;3:1–9.

185. Sharma HC, Srivastava CP, Durairaj C, Gowda CL. Pest management in grain legumes and climate change. In: Yadav SS, McNeil DL, Redden R, Patil SA, editors. Climate Change and Management of Cool Season Grain Legume Crops, Business Media, Springer Science. Dordrecht, The Netherlands, 2010; p. 115–40.

186. Petzoldt C, Seaman A. Climate change effects on insects and pathogens. Climate Change and Agriculture: Promoting practical and profitable responses. 2010. Available from: URL: http://www.panna.org/sites/default/files/CC%20insects&pests.pdf.

187. Fand B, Kamble A, Kumar M. Will climate change pose serious threat to crop pest management: a critical review? International

Journal of Scientific and Research Publications 2012;2(11):1–14. Available from: URL: http://www.ijsrp.org/research-paper-1112/ijsrp-p1109.pdf.

188. Reddy P. Impact of climate change on insect pests, pathogens and nematodes. Pest Management in Horticultural Ecosystems, 2013;19(2):225–33.

189. Hartman GL, Noel GR, Gray LE. Occurrence of soybean sudden death syndrome in east-central Illinois and associated yield losses. Plant Disease 1995;79:314–18.

190. Roy KW, Rupe JC, Hershman DE, Abney TS. Sudden death syndrome. Plant Disease 1997;81:1100–11.

191. Yamamura K, Kiritani K. A simple method to estimate the potential increase in the number of generations under global warming in temperate zones. Applied Entomology and
Q18 Zoology 1998;289–98.

192. Merrill S, Deutsch CA, Tewksbury JJ. Global warming: more insects, eating more crops. Science. 31 August 2018. Available from: URL: http://www.newswise.com/articles/view/699572/?sc=dwtn.

193. Mussolin D. Insects in a warmer world: ecological, physiological and life history responses of true bugs (Heteroptera) to climate change. Global Change Biology 2007;13:1565–85.

194. Deutsch CA, Tewksbury JJ, Huey RB, Sheldon KS, Ghalambor CK, Haak DC, *et al.* Impacts of climate warming on terrestrial ectotherms across latitude. PNAS 2008;105(18):6668–72; https://doi.org/10.1073/pnas.0709472105.

195. Deutsch CA, Tewksbury JJ, Tigchelaar M, Battisti DS, Merrill SC, Huey RB, *et al.* Increase in crop losses to insect pests in a warming climate. Science 2018;361:916–19. 31 August 2018.

196. Chakraborty S. Migrate or evolve: options for plant pathogens under climate change. Global Change Biology 2013;19(7):1985–2000.

197. Chakraborty S, Murray G, White N. Impact of Climate Change on Important Plant Diseases in Australia. A report for the Rural Industries Research and Development Corporation, RIRDC, 2002; Publication No. W02/010, RIRDC Project No. CST-4A.

198. Dalla Pria M, Christiano RCS, Furtado EL, Amorim L, Bergamin Filho A. Effect of temperature and leaf wetness duration on infection of sweet oranges by Asiatic citrus canker. Plant Pathology 2006;55:657–63.

199. Hamilton JG, *et al.* Anthropogenic changes in tropospheric composition increase susceptibility of soybean to insect
Q19 herbivory. Environmental Entomology 2005;34:479–85.

200. Zavala JA, Casteel CL, Nabity PD, Berenbaum MR, DeLucia EH. Role of cysteine proteinase inhibitors in preference of Japanese beetles (*Popillia japonica*) for soybean (*Glycine max*) leaves of different ages and grown under elevated CO_2. Oecologia 2009;161:35–41.

201. DeLucia EH, Casteel CL, Nabity PD, O'Neill BF. Insects take a bigger bite out of plants in warmer, higher carbon dioxide world. Proceedings of the National Academy of Sciences of the United States of America 2008;105:1781–82.

202. Dermody O, O'Neill BF, Zangerl AR, Berenbaum MR, DeLucia EH. Effects of elevated CO_2 and O_3 on leaf damage and insect abundance in a soybean agroecosystem. Arthropod-Plant Interactions 2008;2:125–35.

203. Coll M, Hughes L. Effects of elevated CO_2 on an insect omnivore: a test for nutritional effects mediated by host plants and prey. Agriculture, Ecosystems and Environment 2008;123:271–79.

204. Quarles W. Global warming means more pests. The IPM Practitioner 2007; Vol. XXIX, No. 9/10, September/October.

205. Patterson DT, Westbrook JK, Joyce RJV, Lingren PD, Rogasik J. Weeds, insects, diseases. Climate Change 1999;43(4):711–27.

206. Mattson W, Haack R. The role of drought in outbreaks of plant-eating insects. Biological Sciences 1987;37:374–80.

207. Chen C, McCarl B. An investigation of the relationship between pesticide usage and climate change. Climatic Change 2001;50:475–8.

208. Halpert M, Ropelewski C. North American droughts of the 1980s: A historical perspective. Sixth Conference on Applied Climatology. American Meteorological Society Boston 1989; pp. 88–91. 17.

209. Stinner BR, Taylor RAJ, Hammond RB, Purrington FF, McCartney DA, Rodenhouse N, *et al.* Potential effects of climate change on plant-pest interactions. In: Smith J, Tirpak D editors. The Potential Effects of Global Climate Change in the United States. EPA-230-05-89-053. Appendix C, Agriculture, Vol. 2, Washington, DC; 1989; p. 8-1–8-35.

210. Luck J, Spackman M, Freeman A, Trebicki P, Griffiths W, Finlay K, *et al.* Climate change and diseases of food crops. Plant Pathology 2011;60:113–21.

211. Isard S, Gage S, Comtois P, Russo J. Principles of aerobiology applied to soybean rust as an invasive species. Biological Sciences 2005;55:851–62.

212. Carson RL. Silent Spring. Houghton Mifflin, Boston; 1962. 368 p.

213. Van den Bosch R. The Pesticide Conspiracy. Doubleday, New York; 1978. 212 p.

214. Panizzi AR. History and contemporary perspectives of the integrated pest management of soybean. Neotropical Entomology 2013;42:119–27.

215. Moscardi F, Souza ML. Baculovírus para o controle de pragas: panacéia ou realidade? Biotecnologia Ciência &
Desenvolvimento 2002;24:22–9. Q20

216. Kogan M, Turnipseed SG, Oliveira EB, Borgo A. Pilot insect pest management program for soybean in southern Brazil. Journal of Economic Entomology 1977;70:659–63.

217. Gazzoni DL. Manejo de pragas da soja: uma abordagem histórica. Embrapa, Centro Nacional de Pesquisa de Soja, Embrapa, Londrina, PR, Documentos 78;1994; 72 p.

218. Bueno de Freitas A, Panizzi AR, Spalding Corrêa-Ferreira B, Hoffmann-Campo CB, Ricardo Sosa-Gómez D, Gazzoni DL, *et al.* Histórico e evolução do manejo integrado de pragas da soja no Brasil. In: Hoffmann-Campo CB, Spalding Corrêa-Ferreira B, Moscardi F, editors. Soja-manejo integrado de insetos e outros Artropods-praga. Embrapa, Brasilia; 2012. 859 pp. Available from: URL: http://www.cnpso.embrapa.br/
artropodes/Capitulo0.pdf. Q21

219. Heinrichs EA, da Silva RFP. Estudo de niveis de população de *Anticarsia gemmatalis* Hübner, 1818 e *Plusia* sp. em soja. Agronomia Sulriograndense, Porto Alegre 1975;11:29–35.

220. Heinrichs EA, da Silva RFP. Controle de *Anticarsia gemmatalis* e *Plusia* sp. com insecticidas em po e sua relação com o desfolhomento e rendimento de soja. Anais da Sociedade Entomologica do Brasil 1975;4:78–84.

221. Galileo MHM, de Gastal HAO, Heinrichs EA. Occorencia do fungo, *Nomuraea rileyi* (Farlow) Samson, de taquinideos e himenopteros parasitos em *Anticarsia gemmatalis* Hübner e *Plusia* sp. (Lepidoptera, Noctuidae) criadas em laboratorio. Iheringia, Série Zoologia, Porto Alegre 1997;50:51–9.

222. Allen GE, Knell JD. A nuclear polyhedrosis virus of *Anticarsia gemmatalis*: I. Ultrastructure, replication, and pathogenicity. Florida Entomologist 1997;60:233–40.

223. Carner GR, Turnipseed SG. Potential of a nuclear polyhedrosis virus for the control of the velvetbean caterpillar in soybean. Journal of Economic Entomology 1997;70:608–10.

224. Moscardi F. Utilizacão de *Baculovirus anticarsia* no controle da lagarta da soja, *Anticarsia gemmatalis.* Centro Nacional de Pesquisa de Soja, Embrapa. Londrina, PR, Com. Tec. 23, 1983. 21 p.

225. Moscardi F. Use of viruses for pest control in Brazil: the case of nuclear polyhedrosis virus of the soybean caterpillar, *Anticarsia gemmatalis*. Memórias do Instituto Oswaldo Cruz 1989;84:51–6.

226. Hoffmann-Campo CB, Correa-Ferreira BS, Moscardi F (editors). Soja-manejo integrado de insetos e outros Artropodes-praga. Embrapa 2012. 859 pp. Available from: URL: http://www.cnpso.embrapa.br/artropodes/Capitulo1.pdf.

227. Galileo MHM, Heinrichs EA. Retenção foliar em plantas de soja (*Glycine max* (L.) Merrill) por efeito da ação de *Piezodorus guildinii* (Westwood, 1837) Hemiptera: Pentatomidae), em diferentes ńiveis e épocas de infestaçao. Anais da Sociedade Entomologica Brasileira 1978;7:75–88.

228. Galileo MHM, Heinrichs EA. Avaliação dos danos causados por *Piezodorus guildinii* (Westwood, 1837) (Hemiptera: Pentatomidae), em diferentes ńiveis e épocas de infestação, na qualidade da semente de soja (*Glycine max* (L.). Anais da Sociedade Entomologica Brasileira 1978;7:89–98.

229. Corso IC, Heinrichs EA, Lehman PS. Occorencia do fungo *Nematospora coryli* Peglion em percevejos que atacam a soja. Anais da Sociedade Entomologica do Brazil 1975;4:49–52.

230. Galileo MHM, Heinrichs EA. Efeito dos danos causados por *Piezodorus guildinii* (Westwood, 1837) (Hemiptera: Pentatomidae), em differentes ńiveis e épocas de infestação, no rendimento de graos de soja (*Glycine max* (L.) Merrill). Anais da Sociedade Entomologica Brasileira 1978;7:20–5.

231. Galileo MHM, Heinrichs EA. Avaliação dos danos causados aos sementes de soja (*Glycine max* (L.) Merrill por *Peizodorus guildinii* (Westwood, 1837) (Hemiptera: Pentatomidae) en diferentes ńiveis e épocas de infestação. Anais da Sociedade Entomologica Brasileira 1978;7:40–6.

232. Galileo MHM, Heinrichs EA. Avaliação dos danos causados aos legumes da soja (*Glycine max* (L.) Merrill) por *Piezodorus guildinii* (Westwood, 1837) (Hemiptera: Pentatomidae), em diferentes ńiveis e épocas de infestação. Anais da Sociedade Entomologica Brasileira 1978;7:33–9.

233. Correa-Ferreira BS. Parasitóides de ovos de percevejos: incidência natural, biologia e efeito sobre a população de percevejos da soja [PhD thesis]. Universidade Federal do ParanáCuritiba, Brazil; 1991.

234. Corrêa-Ferreira B.S. Utilização do parasitóide de ovos *Trissolcus basalis* (Wollaston) no controle de percevejos da soja. Londrina: EMBRAPA-CNPSo, 1993. 40 p. (EMBRAPA-CNPSo. Circular Técnica, 11).

235. Correa-Ferreira BS, Moscardi F. Biological control of stink bugs by inoculative releases of *Trissolcus basalis*. Entomologia Experimentalis et Applicata 1996;79:1–7.

236. Corrêa-Ferreira BS. Utilização do parastoide de ovos *Trissolcus basalis* (Wollaston) no controle de percevejos da soja. 1993; EMBRAPA-CNPSo, (EMBRAPA-CNPSo. circular Tecnica, II), Londrina, PR, Brazil.

237. Correa-Ferreira BS, Moscardi F. Seasonal occurrence and host spectrum of egg parasitoids associated with soybean stink bugs. Biological Control 1995;5:96–201.

238. Correa-Ferreira BS, Domit B, Morales L, Guimaraes RC. Integrated soybean pest management in micro river basins in Brazil. Integrated Pest Management Reviews 2000;5:75–80.

239. Corso IC, Gazzoni DL. Sodium chloride: an insecticide enhancer for controlling pentatomids in soybeans. Pesquisa Agropecuária Brasileira 1998;33:1563–71.

240. Godoy CV, de Freitas Bueno, Gazzeiro DLP. Brazilian soybean pest management and threats to its sustainability. Outlooks on Pest Management 2015;26(3):113–7.

241. Bueno AF, Paula-Moraes SV, Gazzoni DL, Pomari AF. Economic thresholds in soybean-integrated pest management: old concepts, current adoption, and adequacy. 2013;42:439–47. Q22

242. Carmo EL, Bueno AF, Bueno RCOF. Pesticide selectivity for the insect egg parasitoid *Telenomus remus*. Biological Control 2010;55:455–64.

243. Lis JA, Becker M, Schaefer CW. Burrower bugs (Cydnidae). In: Schaefer CW, Panizzi AR, editors. Heteroptera of Economic Importance. CRC, Boca Raton; 2000. p. 405–19.

244. Panizzi AR, Niva CC. Overwintering strategy of the brown stink bug in northern Paraná. Pesquisa Agropecuaria Brasileira 1994;29:509–11.

245. Oliveira LJ, Salvadori JR. Rhyzophagous beetles (Coleoptera: Melolonthidae). In: Panizzi AR, Parra JRP, editors. Insect Bioecology and Nutrition for Integrated Pest Management. CRC, Boca Raton; 2012. p. 353–68.

246. Moscardi F. Assessment of the application of baculoviruses for control of Lepidoptera. Annual Review of Entomology 1999;44:257–89.

247. Sosa-Gómez DR, Delpin KE, Moscardi F, Nozaki MH. The impact of fungicides on *Nomuraea rileyi* (Farlow) Samson epizootics and on populations of *Anticarsia gemmatalis* Hübner (Lepidoptera: Noctuidae), on soybean. Neotropical Entomology 2003;32:287–91.

248. Czepak C, Albernaz KC, Vivan LM, Guimarães HO, Carvalhais T. Primeiro registro de ocorrência de *Helicoverpa armigera* (Hübner) (Lepidoptera: Noctuidae) no Brasil. Pesquisa Agropecuária Tropical 2013;43:110–3. DOI: 10.1590/S1983-40632013000100015.

249. Batistela MJ, Bueno AF, Nishikawa MAN, Bueno RCOF, Hidalgo G, Silva L, *et al.* Reevaluation of leaf-lamina consumer thresholds for IPM decisions in short-season soybeans using artificial defoliation. Crop Protection 2012;32:7–11.

250. Bueno Regiane COF, Raetano CG, Dorneles J, Jr Carvalho Fernando K. Integrated pest management of soybean pests: the example of Brazil. Outlooks on Pest Management, August 2017;28(4);149–53(5).

251. Reuters Report. Available from: URL: http://www.reuters.com/investigates/special-report/brazil-pesticides/.

252. Norton GW, Tjornhom J, Bosch D, Ogrodowczyk J, Edwards C, Yamagiwa T, *et al.* Pesticide and IPM policy analysis. In: Norton GW, Heinrichs EA, Luther GC, Irwin ME, editors. Globalizing Integrated Pest Management: A Participatory Research Process. Blackwell Publishing, Ames, Iowa; 2005. p. 191–210.

Author Queries:

Q1: Please check whether ‘IPM’ should be expanded as ‘Integrated pest management’ in the article title.
Q2: Please confirm the expansion introduced for ‘IPM’.
Q3: Please provide ‘Keywords’ for this paper.
Q4: Please confirm the set heading levels throughout the paper.
Q5: Both ‘soyabean’ and ‘soybean’ are used in this paper. Please make it consistent with either of the spelling.
Q6: In the sentence beginning “In 2006 and 2007…” should “Munkyung City” be changed to “Mungyeong City”?
Q7: We have changed ‘fall’ to ‘autumn’ four times in this paper. Please confirm.
Q8: In the sentence beginning “Among the various isolates…” should “*Scelerotia*” be changed to “*Sclerotia*”?
Q9: Please check the edits made to the sentence beginning ‘The change was dramatic...’.
Q10: Please check references are renumbered appearance wise.
Q11: Please provide page range in ref. [17].
Q12: Please provide publisher location in ref. [26].
Q13: Please provide initial for editor ‘Mota-Sanchez’ in ref. [39].
Q14: Please provide Institution and publisher location in ref. [57].
Q15: Please confirm the journal title ‘Anais da Sociedade Entomológica do Brasil’ in ref. [61].
Q16: Please confirm the journal title ‘Phytopathologische Zeitschrift’ in ref. [96].
Q17: Please provide volume number and page range in ref. [100].
Q18: Please provide volume number in ref. [191].
Q19: As per journal style, names of up to six authors are provided. If there are more than six, only the first six should be given followed by *et al*. Please provide next 5 author names in ref. [199] as required by journal style.
Q20: Please confirm the journal title ‘Biotecnologia Ciência & Desenvolvimento’ in ref. [215].
Q21: Please provide publisher location in ref. [218].
Q22: Please provide journal title for ref. [241].
Q23: Please check that ‘*Melancanthus scutellaris*’ has been displayed correctly.