INDUSTRY BIOSECURITY PLAN

FOR THE GRAINS INDUSTRY

Generic Contingency Plan

Sap-sucking insect transmitted viruses affecting the grains industry

Specific examples detailed in this plan:

***Peanut Stripe Virus* (*Potyvirus*),**

***Chickpea Chlorotic Dwarf Virus* (*Geminivirus*)**

and

***Chickpea Chlorotic Stunt Virus* (*Polerovirus*)**

Plant Health Australia

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Plant Health AUSTRALIA

GRDC Grains Research & Development Corporation

CRC PLANT biosecurity

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1 Purpose and background of this contingency plan

Developing a pest contingency plan for groups of exotic pests will ensure the industry is prepared for a wider range of new pest incursions. These broader focused contingency plans are designed to assist the grains industry during an incursion of a sap-sucking insect transmitted virus that may not already be covered by a pest specific contingency plan. As sap-sucking insect transmitted viruses employ the same transmission pathways (i.e. sap-sucking insects that transfer (vector) the virus between host plants, and may or may not be seed-borne) and control options (i.e. insecticides to control their vectors, destruction of infected plants, restrictions on the movement of seed (if seed-borne)) this contingency plan provides information for the management of various sap-sucking insect transmitted viruses.

This contingency plan provides background information on the biology of the pest available control measures and other relevant information to assist with preparing for and responding to an incursion into Australia of viruses that are transmitted by aphids, leafhoppers (Jassids) and other sap-sucking insects that could potentially impact on the grains industry. In this contingency plan three sap-sucking insect transmitted viruses have been used as examples of exotic viruses that could potentially enter Australia with infected insect vectors or infected plants and seed from overseas.

The contingency plan provides guidelines and options to be considered when developing a Response Plan for an incursion of an exotic insect transmitted plant virus. Any Response Plan developed using information in whole or in part from this contingency plan must follow procedures as set out in PLANTPLAN and be endorsed by the National Management Group prior to implementation.

The information for this plan has been primarily obtained from documents as cited in the reference section. Information on the background, life cycle, host range, distribution and symptoms of three specific viruses are given are given as examples, with the emphasis of this document on the management options in the event of a sap-sucking insect transmitted virus incursion in Australia

2 Australian grains industry

The grains industry is the largest plant industry in Australia and grain crops are grown in all states and territories. The grains industry is primarily situated in a narrow crescent running through the mainland states, known as the grain belt. This area stretches from central Queensland, through New South Wales, Victoria and southern South Australia. In Western Australia, the grain belt covers the south-west corner of the state. Wheat is the most widely planted grain and is grown in all areas of the grain belt (Figure 1).

The grains industry consists of 25 leviable crops; many are affected by sap-sucking insect transmitted viruses.

Due to Australia's relatively small population and domestic demand, export markets are essential for the viability of Australian grain farms. Australia is one of the world's largest grain exporters, exporting millions of tonnes of grain annually. With this reliance on exports, maintaining our current plant health status through appropriate biosecurity measures is of utmost importance in retaining access to these markets.

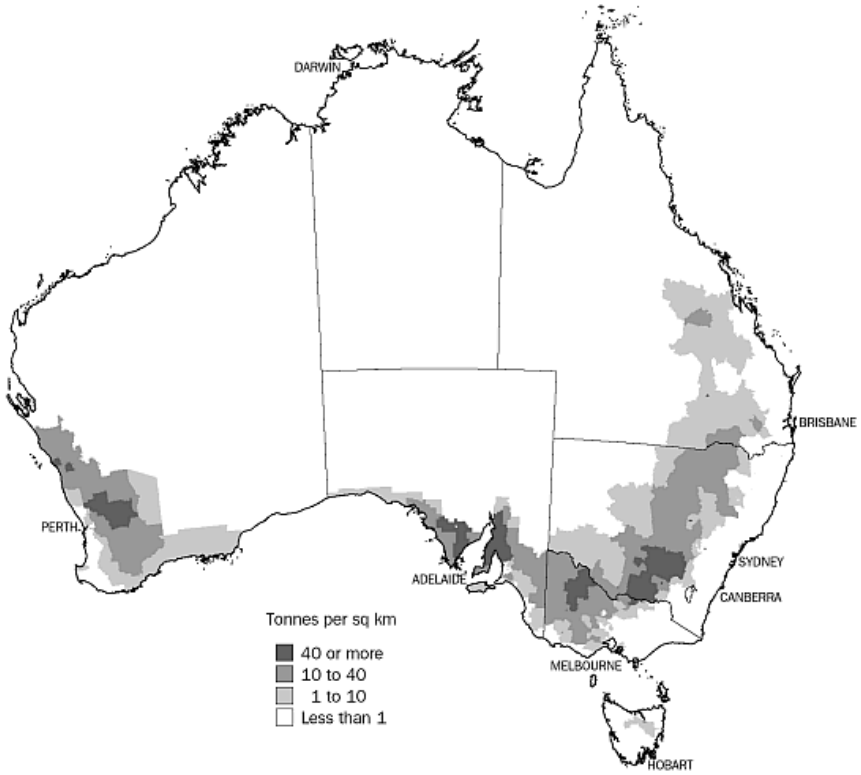

Figure 1 *Map of wheat producing regions in Australia (i.e. the grain belt). (Source ABS 2007)*

2.1 Notification process for the reporting of suspect pests

Early detection and reporting may prevent or minimise the long-term impact of an incursion into Australia of a sap-sucking insect transmitted virus. The notification process is described in Figure 2.

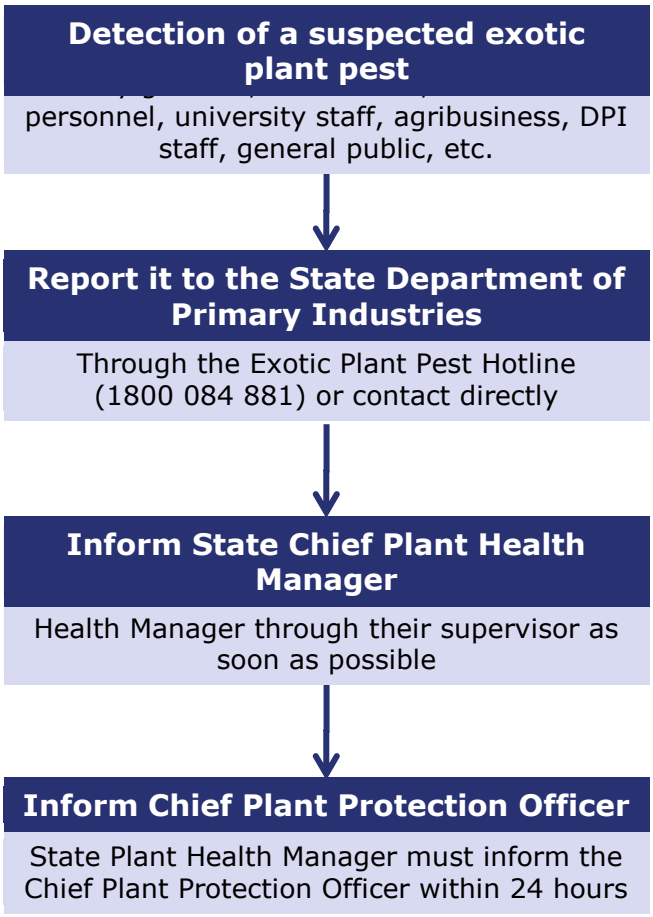

Figure 2. *Notification process for the reporting of suspect pests*

3 Eradication or containment decision matrix

The decision to eradicate should be based on the potential economic impact of host damage resulting from the pest(s), the cost of eradication and technical feasibility. Eradication costs must factor in long term surveys to prove the success of the eradication program. A minimum of three years with no detection of the pathogen may be necessary before pest free status can be declared. The exact time required will depend on the survival ability of the specific pathogen in the absence of host plants or sap-sucking insect vector.

No specific eradication matrix has been determined for any sap-sucking insect transmitted viruses, however the key decision points during the Investigation and Alert Phase are outlined in PLANTPLAN and Table 2 should be followed in determining if an incursion of this pest will result in eradication or management/containment. The final decision between eradication and management will be made through the National Management Group.

Table 1. *Factors considered in determining whether eradication or alternative action will be taken for an EPP Incident (taken from: Table 2; Section 4.16 of PLANTPLAN)*

Factors favouring eradication	Factors favouring alternative action
• Cost/benefit analysis shows significant economic loss to industry or the community if the organism established • Physical barriers and/or discontinuity of host between production districts. • The generation time, population dynamics and dispersal of the organism favour more restricted spread and distribution. • Vectors discontinuous in distribution and can be effectively controlled. • Outbreaks few and confined. • Trace back information indicates few opportunities for secondary spread. • Weather records show unfavourable conditions for pest development. • Ease of access to outbreak site and location of alternate hosts. • Pathways for reintroduction from international trade closed.	• Cost/benefit analysis shows relatively low economic or environmental impact if the organism establishes. • Major areas of continuous production of host plants. • Short generation times, potential for rapid population growth and long distance dispersal lead to rapid establishment and spread. • Vectors unknown, continuous in distribution or difficult to control. • Outbreaks numerous and widely dispersed. • Trace back information indicates extensive opportunities for secondary spread. • Weather records show optimum conditions for pest development. • Terrain difficult and/or problems accessing and locating host plants. • Pathways for reintroduction from international trade open.

4 General information on Sap-sucking insect transmitted viruses

4.1 Exotic sap-sucking insect transmitted viruses affecting the grains industry

There are 92 viruses identified in the Industry Biosecurity Plan (IBP) for the grains industry (Plant Health Australia 2009-review 2014). Approximately 60 of these are vectored by sap-sucking insects. Three of these viruses are used in this contingency plan as examples of sap-sucking insect transmitted viruses. Although the specific controls (e.g. the active ingredients of any pesticides used to control the insect vectors, etc.) will vary between viruses and vectors, the management of most exotic insect transmitted viruses will be similar. For example the general procedures for control (Section 7.3.1), sampling protocols (Section 7.2), quarantine and movement control (Section 8.3), zoning requirements (Section 8.4) and other components of this contingency plan will be the same for most insect transmitted viruses.

There are more than 60 exotic sap-sucking insect transmitted viruses that have been identified in the Grains IBP (Plant Health Australia 2009-review 2014). The economic impact and overall risk posed by these viruses, their hosts, vectors and ability to be transmitted with seed is summarised in Table 2.

Table 2 *Exotic sap-sucking insect transmitted viruses identified in the grains Industry Biosecurity plan (Plant Health Australia* 2009-review 2014*)*

Virus	Primary (major) host	Secondary (minor) hosts	Overall risk	Main vectors of disease	Are vectors endemic or exotic	Is virus seed transmitted
Agropyron mosaic virus (Rymovirus)	Wheat, rye, barley and Quack grass (*Agropyron repens*)	-	NEGLIGIBLE	Cereal rust mite (*Abacarus hystrix*)[1]	Endemic	Not seed-borne
Barley yellow striate mosaic virus (Cytorhabdovirus)	Barley, wheat, oats	-	NEGLIGIBLE (with or without vector)	Leafhopper (*Laodelphax striatellus*)	Exotic	Not seed-borne
Bean common mosaic virus (Potyvirus), peanut stripe strain[2]	Peanuts, soybean, cowpea	-	HIGH	Aphids including (*Aphis craccivora, A. gossypii, A. glycines, Hysteroneura setariae, Myzus persicae*)	Endemic	Seed borne only on peanut.
Bean golden mosaic virus (Begomovirus)	Soybean, common bean, lima bean	Mungbean, cowpea, chickpea, peanut, field pea, lentil	LOW-VERY LOW	Silverleaf whitefly (*Bemisia tabaci*)	Endemic	Not seed-borne
Bean yellow dwarf virus (Mastrevirus)	Common bean, chickpea	-	UNKNOWN	Unknown leafhopper species	Unknown	Not seed-borne
Bean yellow vein banding virus (Umbravirus)	Faba bean, common bean	-	NEGLIGIBLE	Aphids (*Acyrthosiphon pisum and Myzus persicae*)	Endemic	Not seed-borne
Bidens mottle virus (Potyvirus)	Narrow leaf lupin, ornamental plants including: black eyed Susan, zinnia	-	VERY LOW	Aphids (*Aphis craccivora, A. spiraecola, Acyrthosiphon pisum, Lipaphis erysimi* and *Myzus persicae*)	Endemic	Not seed-borne
Brazilian wheat spike virus (Tenuivirus)	Wheat	-	UNKNOWN	Unknown Delphacid plant hoppers	Unknown	Not seed-borne
Broad bean severe chlorosis virus (Unclassified)	Faba bean	-	VERY LOW	Pea aphid (*Acyrthosiphon pisum*)	Endemic	Not seed-borne

[1] Arachnid rather than an insect but virus would be managed in the same manner as sap-sucking insect transmitted viruses.

[2] One of the examples used in this contingency plan.

Virus	Primary (major) host	Secondary (minor) hosts	Overall risk	Main vectors of disease	Are vectors endemic or exotic	Is virus seed transmitted
Chickpea bushy dwarf virus (Potyvirus)	Chickpea	-	VERY LOW-NEGLIGIBLE	Unknown aphid	Unknown	Not seed-borne
Chickpea chlorotic dwarf Syria (Mastrevirus)	Chickpea	-	LOW	Leafhopper: (*Orosius albicinctus*)	Exotic	Not seed-borne
Chickpea chlorotic dwarf virus (Mastrevirus) (syn. Chickpea chlorotic dwarf virus (Geminivirus))[3]	Chickpea, faba bean, field pea, lentil, tobacco, tomato, sugar beet	-	MEDIUM	Leafhoppers: (*Orosius orientalis, O. albicinctus* and *Neolimnus aegyptiacus*)	*O. albicinctus* and *Neolimnus aegyptiacus* exotic. *O. orientalis* is endemic	Not seed-borne
Chickpea chlorotic stunt virus (Polerovirus)[2]	Chickpea, faba bean, field pea, lentil, vetch, medics and some other legumes	-	MEDIUM	Aphids (*Aphis craccivora, Acyrthosiphon pisum*)	Endemic	Not seed-borne
Chickpea distortion mosaic virus (Potyvirus)	Chickpea	-	VERY LOW - NEGLIGIBLE	Cotton aphid (*Aphis gossypii*)	Endemic	Not seed-borne
Chickpea filiform virus (Potyvirus)	Chickpea	-	LOW-NEGLIGIBLE	Aphids (*Acyrthosiphon pisum, Myzus persicae*)	Endemic	Not seed-borne
Chickpea yellows virus (Luteovirus)	Chickpea	-	UNKNOWN	Unknown aphids	Unknown	Not seed-borne
Clover yellows virus (Closterovirus)	Faba bean and *Trifolium* spp.	-	VERY LOW	Cowpea aphid (*Aphis craccivora*)	Endemic	Not seed-borne
Cowpea mild mottle virus (Carlavirus)	Peanut, soybean, common bean, tomato, cowpea	-	VERY LOW	Silver leaf whitefly (*Bemisia tabaci*)	Endemic	Not seed-borne
Cynodon chlorotic streak virus (Nucleorhabdovirus)	Bermuda blue grass, maize	-	LOW-NEGLIGIBLE (with or without vector)	Leafhopper (*Toya propinqua*)	Exotic	Not seed-borne
Faba bean necrotic stunt virus (Nanovirus)	Chickpea, field pea, faba bean, soybean, cowpea and common beans	-	LOW	Aphids: (*Acyrthosiphon pisum, Aphis craccivora*)	Endemic	Not seed-borne

[3] One of the examples used in this contingency plan.

Virus	Primary (major) host	Secondary (minor) hosts	Overall risk	Main vectors of disease	Are vectors endemic or exotic	Is virus seed transmitted
Faba bean necrotic yellows virus (Nanovirus)	Chickpea, *Lathyrus* spp., lentil, field pea, faba bean, common vetch, common bean, bur clover, *Trifolium* spp., cowpea	-	LOW	Aphids (*Acyrthosiphon pisum and Aphis craccivora, A. fabae*)	*Acyrthosiphon pisum* and *A. craccivor* endemic. *A. fabae* exotic	Not seed-borne
Groundnut bud necrosis virus (Tospovirus)	Peanut, field pea, cowpea, mungbean, soybean	-	HIGH	Melon thrips (*Thrips palmi*)	Restricted distribution	Not seed-borne
Groundnut ringspot virus (Tospovirus)	Peanut, soybean, tomato, capsicum, tomatillo, coriander	-	MEDIUM	Thrips including: *Frankliniella occidentalis, F. schultzei, F. gemina* (McAvoy et al 2011)	*Frankliniella occidentalis* and *F. schultzei* endemic. *F. gemina* exotic	Not seed-borne
Guinea grass mosaic virus (Potyvirus)	Guinea grass, maize, pearl millet	-	MEDIUM-NEGLIGIBLE	Corn aphid (*Rholpalosiphum maidis*) (Lamy et al., 1979)	Endemic	Not seed-borne
Iranian wheat stripe virus (Tenuivirus)	Wheat, barley, oat, rice, rye, sorghum	-	VERY LOW-LOW (with or without vector)	Leafhopper (*Unkanodes tanasijevici*)	Exotic	Not seed-borne
Lentil stunt virus (Polerovirus)	Lentil	-	UNKNOWN	Unknown aphid	N/A	Not seed-borne
Lettuce infectious yellows virus (Crinivirus)	Lettuce, melon, pumpkin, sugar beet, sunflower, sow thistle, dandelion	-	VERY LOW	Silver leaf whitefly (*Bemisia tabaci*)	Endemic	Not seed-borne
Lupin leaf curl virus (Begomovirus)	Lupin	-	VERY LOW	Silver leaf whitefly (*Bemisia tabaci*)	Endemic	Not seed-borne
Maize chlorotic dwarf virus (Waikavirus)	Johnson grass, maize, sorghum	-	LOW (without vector) MEDIUM (with vector)	Leafhoppers (*Graminella nigrifrons, G. sonora* and *Exitianus exitiosus*)	Exotic	Not seed-borne
Maize gooseneck stripe virus (Unclassified)	Maize	-	MEDIUM-NEGLIGIBLE	Leafhopper (*Peregrinus maidis*)	Endemic	Not seed-borne

Virus	Primary (major) host	Secondary (minor) hosts	Overall risk	Main vectors of disease	Are vectors endemic or exotic	Is virus seed transmitted
Maize Iranian mosaic virus (Nucleorhabdovirus)	Maize	Reported to infect wheat and barley (Ammar et al., 2004))	MEDIUM-NEGLIGIBLE	Leafhoppers (*Ribautodelphax notabilis* and *Peregrinus maidis*) (Ammar et al., 2004)	*Peregrinus maidis* is endemic. *Ribautodelpha x notabilis* is exotic	Not seed-borne
Maize line virus (Unclassified)	Maize	-	MEDIUM-NEGLIGIBLE	Leafhopper (*Peregrinus maidis*)	Endemic	Not seed-borne
Maize mosaic virus (Nucleorhabdovirus)	Maize, sorghum	-	LOW	Leafhopper (*Peregrinus maidis*)	Endemic	Not seed-borne
Maize mottle virus (Begomovirus) (syn. Chlorotic stunt virus)	Maize	-	LOW-NEGLIGIBLE (with or without vector)	Leafhoppers (*Cicadulina mbila*, *C. zeae, C. storeyi* (syn. *C. triangula*)) (Irwin et al., 1999)	Exotic	Not seed-borne
Maize raya gruesa virus (Unclassified)	Maize	-	MEDIUM-NEGLIGIBLE	Leafhopper (*Peregrinus maidis*)	Endemic	Not seed-borne
Maize rayado fino virus (Marafivirus)	Maize, teosinte	-	VERY LOW-LOW (with or without presence of vectors)	Leafhopper (*Dalbulus maidis*)	Exotic	Not seed-borne
Maize rough dwarf virus (Fijivirus)	Maize	Barley, oats, wheat, sorghum	VERY LOW (with or without presence of vectors)	Leafhopper (*Laodelphax striatellus*)	Exotic	Not seed-borne
Maize streak virus (Mastrevirus)	Maize, sugarcane, millet, sorghum	Wheat, barley, oats, rye, rice, finger millet, pearl millet	LOW (with or without presence of vectors)	Leafhoppers in the *Cicadulina* genus including: *C. mbila* (main vector), *C. storeyi, C. arachidis, C. similis, C. ghaurii*	Exotic	Not seed-borne
Milk vetch dwarf virus (Luteovirus)	Field pea, faba bean, milk vetch, soybean, cowpea, common bean	-	VERY LOW	Cowpea aphid (*Aphis craccivora*)	Endemic	Not seed-borne

Virus	Primary (major) host	Secondary (minor) hosts	Overall risk	Main vectors of disease	Are vectors endemic or exotic	Is virus seed transmitted
Mungbean yellow mosaic virus, Mungbean yellow mosaic India virus, Dolichos yellow mosaic virus and Horsegram yellow mosaic virus (Begomovirus)	Lima bean, mungbean, cowpea, black gram, pigeon pea, common bean, lablab, soybean	-	MEDIUM	Silver leaf whitefly (*Bemisia tabaci*)	Endemic	Not seed-borne
Oat blue dwarf virus (Marafivirus)	Oats, barley, flax, wheat	-	NEGLIGIBLE (with or without vectors)	Aster leafhopper (*Macrosteles quadrilineatus*)	Exotic	Not seed-borne
Oat sterile dwarf virus (Fijivirus)	Oats, barley, wheat, meadow fescue and to Italian and perennial ryegrasses	-	LOW (with or without vectors)	Plant hoppers (*Javesella pellucida* and *Dicranotropis hamata*)	Exotic	Not seed-borne
Pea enation mosaic virus (Enamovirus + Umbravirus)	Chickpea, faba bean, narrow leaf lupin, field pea, lentils, common bean, grass pea, lucerne, clovers, medics and sweet pea, vetch, crimson clover	-	VERY LOW	Aphids: (*Acyrthosiphon pisum, Aphis frangulae, Aulacorthum circumflexum, A. solani, Sitobion avenae, Macrosiphum euphorbiae, Megoura viciae, Myzus persicae, Rhopalosiphum padi, Schizaphis graminium*)	*Acyrthosiphon pisum, Aulacorthum circumflexum, A. solani, Macrosiphum euphorbiae, Myzus persicae, Rhopalosiphum padi* are endemic. *Aphis frangulae, Sitobion avenae, Megoura viciae, Schizaphis graminium* are Exotic	Not seed-borne
Pea necrotic yellow dwarf virus (Nanovirus)	Field pea	-	MEDIUM-NEGLIGIBLE	Pea aphid (*Acyrthosiphon pisum*)	Endemic	Not seed-borne
Pea streak virus (Carlavirus)	Field pea, chickpea, lentil, faba bean, lucerne, red clover, white clover, white sweet clover	-	LOW	Pea aphid (*Acyrthosiphon pisum*)	Endemic	Not seed-borne

Virus	Primary (major) host	Secondary (minor) hosts	Overall risk	Main vectors of disease	Are vectors endemic or exotic	Is virus seed transmitted
Peanut stunt virus (Cucumovirus)	Peanut, field pea, lupin, soybean, clovers, lucerne	-	LOW	Aphids (*Aphis craccivora, A. spiraecola*, *Myzus persicae*)	Endemic	Seed-borne on soybean and peanut
Plum pox virus (Potyvirus)	Lupin, Prunus spp., vetch, clovers and medics	-	VERY LOW	Aphids (*Aphis craccivora, Brachycaudus cardui* (plum and apricot only), *B. helichrysi, Myzus persicae, Phorodon pruni* (syn. *Phorodon humuli*))	*Aphis craccivora*, *B. helichrysi, Myzus persicae* are endemic. *Brachycaudus cardui, Phorodon pruni* are exotic	Not seed-borne
Red clover vein mosaic virus (Carlavirus)	Chickpea, lentil, faba bean, sweet pea, field pea, common bean, lucerne, Melilotus spp., Trifolium spp.	-	MEDIUM	Aphids (*Acyrthosiphon pisum, Aphis fabae, Cavariella aegopodii, C. theobaldi* (affects willow and Umbelliferae), *Myzocallis onomidis, Myzus persicae, Therioaphis maculata*)	*Acyrthosiphon pisum, Cavariella aegopodii, Myzus persicae Therioaphis maculata* (syn. *Therioaphis trifolii*) are endemic. *Aphis fabae, C. theobaldi, Myzocallis onomidis* and are exotic	Seed borne on faba bean and field pea
Rice black streaked dwarf virus (Fijivirus)	Rice, maize, oats, barley, wheat	-	VERY LOW (with or without vector)	Plant hoppers (*Laodelphax striatellus, Unkanodes sapporona, U. albifascia*)	Exotic	Not seed-borne
Rice hoja blanca virus (Tenuivirus)	Rice	Wheat, oats, rye, barley	NEGLIGIBLE (with or without vector)	Plant hoppers (*Tagosodes cubanus* (syn. *Sogata cubana*) (rice only), *Tagosodes orizicolus* (syn. *Sogata orizicola*))	Exotic	Not seed-borne
Rice stripe virus (Tenuivirus)	Rice, wheat, barley, oats, rye, foxtail millet	-	NEGLIGIBLE (with or without vector)	Plant hoppers (*Laodelphax striatellus, Unkanodes sapporona, U. albifascia*, *Terthron albovittatus* (rice only))	Exotic	Not seed-borne

Virus	Primary (major) host	Secondary (minor) hosts	Overall risk	Main vectors of disease	Are vectors endemic or exotic	Is virus seed transmitted
Rosette disease (Complex infection of: Groundnut rosette (Umbravirus), Groundnut assister (Luteovirus)[4]	Peanuts	-	VERY LOW	Cowpea aphid (*Aphis craccivora*)	Endemic	Not seed-borne
Russian winter wheat mosaic virus (Rhabdovirus)	Wheat, oats, barley, rye	-	VERY LOW (with or without vector)	Plant hoppers (*Psammotettix striatus*, and *Macrosteles laevis*)	Exotic	Not seed-borne
Sorghum mosaic virus (Potyvirus)	Sorghum, sugarcane	-	LOW (With or without vector)	Brown ambrosia aphid (*Uroleucon ambrosiae* (syn. *Dactynotus ambrosiae*)) (Shukla et al., 1998)	Exotic	Not seed-borne
Sorghum stunt mosaic virus (Rhabdovirus)	Sorghum, maize	The virus has been experimentally transferred to wheat (Creamer et al., 1997)	VERY LOW (with or without vector)	Lesser lawn leafhopper (*Graminella sonora*)	Exotic	Not seed-borne
Sunflower mosaic virus (Potyvirus)	Asteraceae: *Helianthus* spp. (including sunflower), *Sanvitalia* spp., and *Zinnia* spp.	-	LOW	Aphids (*Myzus persicae* and *Capitphorus elaegni*)	Endemic	Seed-borne on sunflower
Sunflower yellow blotch virus (Umbravirus)	Sunflower, peanut, Tridax procumbens	-	MEDIUM-NEGLIGIBLE	Cotton aphid (*Aphis gossypii*) (Kaitisha 2003)	Endemic	Not known if seed-borne
Wheat American striate mosaic virus (Cytorhabdovirus)	Wheat, Durum, *Triticum* spp.	-	VERY LOW (regardless of presence of vector)	Leafhoppers (*Endria inimica* and *Elymana virescens*)	Exotic	Not seed-borne

[4] There are three forms of the virus: Chlorotic rosette, Green rosette and Mosaic rosette.

Virus	Primary (major) host	Secondary (minor) hosts	Overall risk	Main vectors of disease	Are vectors endemic or exotic	Is virus seed transmitted
Wheat dwarf virus (Mastrevirus)	Barley strain infects: barley, oats, maize, triticale. Wheat strain infects wheat and triticale (Mehner et al., 2003). Rye, wheat and triticale were infected in Poland (Jezewska 2001) suggesting wheat strain also affects rye.	-	NEGLIGIBLE (with or without presence of vectors)	Leafhopper (*Psammotettix striatus* (syn. *Psammotettix alienus*))	Exotic	Not seed-borne
Wheat yellow leaf virus (Closterovirus)	Wheat, barley	-	LOW	Aphids (*Rhopalosiphum maidis, R. padi*)	Endemic	Not seed-borne
Zea mosaic virus (Potyvirus)	Maize, sorghum, Johnston grass	-	MEDIUM	Aphids (*Myzus persicae* and *R padi*)	Endemic	Not seed-borne

4.2 General information on lifecycles and dispersal

All plant viruses need access to a living organism to reproduce. Viruses can infect plants utilising a number of different pathways. These include:

- seed transmission (where the virus is able to infect seeds from infected plants and therefore infect plants grown from infected seeds)
- the movement of vectors between infected and non-infected plants (see Section 4.3 for further information on plant vectors)
- the transmission of sap between plants (so could be spread by grafting and similar practices)

A summary of the vectors and seed transmission of the sap-sucking insect transmitted viruses that affect the grains industry is provided in Table 2.

4.3 Background information: Sap-sucking insect vectors

Many viruses are spread, at least in part, by sap-sucking insects including aphids and leafhoppers (jassids). These include viruses from the families Closteroviridae, Geminiviridae, Luteoviridae, Potyviridae, Nanoviridae, Reoviridae, Rhabdoviridae, Secoviridae and Tymoviridae. The family Potyviridae contains the majority of insect transmitted viruses, predominantly in the genus *Potyvirus*.

Over 200 aphid species act as vectors of plant viruses, accounting for around half of insect-vectored viruses (Nault 1997). Other invertebrates can also act as vectors of plant viruses, for example there are five genera of thrips (order: Thysanoptera) that are known to transmit viruses, these are: *Thrips* spp., *Frankliniella* spp., *Scirtothrips* spp., *Microcephalothrips* spp. and *Ceratothripoides* spp. (Jones 2005). Mites (class Arachnidia) from the families Eriophyidae and Tetranychidae can also vector some plant viruses (Slykhuis 1965), as can some nematodes (Brunt 1966). Table 2 provides a summary of the main vectors of each of the 60+ exotic insect transmitted viruses that have been identified in the grains IBP (Plant Health Australia 2009-review 2014). In many cases one or more of the insect vectors of the virus are present in Australia, which would likely have an impact of the rate of spread of the virus once it has enters the country.

The presence of endemic vectors and the ability of the virus to be spread with seed has implications for the management and possible entry pathways for the virus. For example if seed-borne seed would provide an entry pathway into the country and a means of rapid spread between areas within the country.

4.3.1 Virus retention in vectors

Typical of many sap-sucking insect transmitted viruses, *Potyvirus* viruses are transmitted mechanically by the mouthparts of sap-sucking insects, such as aphids, and are non-persistent and non-circulative (Danci et al., 2009). Other viruses such as the *Polerovirus*, Chickpea chlorotic stunt virus, are transmitted in a persistent and circulative manner (Abraham et al., 2006) but are non-propagative (Knierim et al., 2010). The *Geminivirus*, Chickpea chlorotic dwarf virus, is also transmitted in a persistent (Horn et al., 1994), circulative and non-propagative manner (Abraham et al., 2010).

The term non-circulative refers to viruses that are carried by the vector externally or on cells of the animal's stylet or foregut (Gray and Banerjee 1999). This means that non-circulative viruses have a relatively short retention time on or within the vector (Brault *et al*. 2010) and that the vectors only remain viruliferous (i.e. able to carry and infect plants with the virus) for a short period of time (often only minutes to hours (Ng and Perry 2004)). Viruses are termed as being circulative when the virus is retained inside the vector (Gray and Banerjee 1999). Circulative viruses can be further classified as being either propagative or non-propagative depending on the virus's ability to replicate within the vector. Propagative viruses are able to replicate within the vector and some suggest (e.g. Power 2000) that this is because propagative viruses evolved on insects and then became plant viruses at a later date.

Non-persistent viruses are characterised by short acquisition periods (seconds) and short inoculation periods (minutes), while semi-persistent transmission means that the virus has a longer acquisition and retention (acquisition over minutes to hours, and retention of the virus by the vector for hours (Gray and Banerjee 1999; Brault et al., 2010)). Persistent viruses have longer acquisition periods, having acquisition periods of minutes to hours. The virus is then retained for the rest of insect's life (weeks-months).

Most insect transmitted viruses are non-circulative and non-persistent, for this reason, vectors can normally only carry the virus short distances for short periods of time; however, if strong winds are present then the insects can still transmit such viruses over large distances.

4.4 Management implications

The way that the virus is retained by the insect vector can determine how the virus is likely to enter the country and spread between hosts.

For example, if the virus is non-persistent and non-circulative it is unlikely to be introduced on a vector as vectors of non-persistent viruses do not retain the virus for long enough (often only minutes to hours (Ng and Perry 2004)) to enter the country and infect host plants. Because of this non-persistent viruses are more likely to enter the country on infected seed (if seed-borne) or live infected plants. Viruses retained by the vector in a persistent manner could be introduced on an infected vector from overseas as viruses that are retained in a persistent manner are retained by the vector for its life, meaning it is possible that vectors could enter the country carrying a virus and be able to spread the virus to suitable host plants. Any viruses type could also be introduced through infected plant material, such as seed (if seed-borne) or live plants.

The presence of endemic vectors and the ability of the virus to be spread with seed has implications for the management and potential entry pathways for the virus. For example, if seed-borne, seed would provide an entry pathway for the virus into the country and a means of rapid spread between plant hosts.

Regardless of the retention of the virus, or the seed transmission of the virus, it is critical that both the host plant(s) and the sap-sucking insect vector(s) are controlled to slow the spread of the virus and give the best possible chance of eradication.

4.5 General diagnostic information for plant viruses

Viruses can be identified using a number of different techniques these include the use of:

- Electron microscopy – sap extracts are examined for characteristic particle sizes and shapes, which can be used in conjunction with other tests for the diagnosis of the virus causing the disease.
- Enzyme Linked Immunosorbent Assay (ELISA) – a rapid test that can be used to diagnose the presence of specific plant viruses. ELISA is a useful method for large scale testing of material for the presence or absence of plant viruses.
- Polymerase Chain Reaction (PCR) and Reverse Transcription Polymerase Chain Reaction (RT-PCR) – a molecular test that allows rapid, specific, and sensitive test that can be used to detect and diagnose plant viruses.

5 Pest information/status – Sap-sucking insect transmitted viruses

5.1 Example viruses and vectors

Many viruses are vectored by sap-sucking insects. This contingency plan uses three viruses as examples of sap-sucking insect vectored plant viruses that affect grain crops. These are:

- *Peanut stripe virus* (*Potyvirus*)
- *Chickpea chlorotic dwarf virus* (*Geminivirus*)
- *Chickpea chlorotic stunt virus* (*Polerovirus*)

Table 3 lists the main hosts and vectors that are known to transmit these three viruses. specific information on the viruses is provided in Section 5, and information on the vectors in provided in Section 6.

Table 3. *Hosts and vectors of Peanut stripe virus, Chickpea chlorotic dwarf virus and Chickpea chlorotic stunt virus*

Virus	Virus host(s)	Transmission pathways/vector(s)	Reference	Is the vector in Australia
Peanut stripe virus	Peanut; Soybean; Cowpea; See Section 5.2.3 for more details	Seed-borne	Zettler et al., 1993	N/A
		Cowpea aphid (*Aphis craccivora*)	Sreenivasulu and Demski (1988); Adalla and Natural (1988); Choopanya and Kittipakorn (1989)	In Australia
		Green peach aphid (*Myzus persicae*)	Sreenivasulu and Demski (1988)	In Australia
		Cotton aphid (*Aphis gossypii*)	Adalla and Natural (1988); Choopanya and Kittipakorn (1989)	In Australia
		Soybean aphid (*Aphis glycines*)	Choopanya and Kittipakorn (1989)	In southern Queensland and northern NSW
		Rusty plum aphid (*Hysteroneura setariae*)	Saleh et al., (1989)	In Australia
Chickpea chlorotic dwarf virus	Chickpea; Faba bean; Sugar beet; See Section 5.3.3 for more details	Common brown leafhopper (*Orosius orientalis*)	Farzadfar et al., (2008); Horn et al., (1994)	In Australia
		Orosius albicinctus	Akhtar et al., (2011)	Not reported from Australia
		Neolimnus aegyptiacus	Hamed and Makkouk (2002)	Not reported from Australia
Chickpea chlorotic stunt virus	Chickpea; Faba bean; See Section 5.4.3 for more details	Cowpea aphid (*Aphis craccivora*)	Abraham et al., (2006); Asaad et al., (2009)	In Australia
		Pea aphid (*Acyrthosiphon pisum*)	Asaad et al., (2009)	In Australia

5.2 Pest Details – Example 1: Peanut Stripe Virus

Common name:	Peanut Stripe Virus (PStV)
Scientific name:	*Peanut Stripe Virus* (*Potyvirus*)
Synonyms:	Groundnut stripe disease; Groundnut mosaic virus; Peanut mild mottle virus; Peanut blotch virus; Peanut chlorotic ringspot virus; *Bean Common Mosaic Potyvirus* - strain: *Peanut Stripe Virus.*
Taxonomic position:	Group: Group IV (Positive sense ssRNA virus) Family: Potyviridae Genus: *Potyvirus*

The information from this plan has been primarily obtained from documents as cited in the reference section as well as material sourced from the 'Pest Risk Review for Peanut stripe virus (PStV) (Coutts 2005).

5.2.1 Background

Peanut stripe virus (PStV) causes significant losses to a number of crops, including peanuts and soybeans (Zettler et al., 1993). Yield losses in infected crops in Indonesia reach 30 to 40% (Saleh et al., 1992), 23% yield losses have been reported in China (Xu et al., 1983) and yield losses of 21 - 23% have been reported in the USA (Demski et al.,1984).

Peanut Stripe Virus is a member of the genus *Potyvirus* and the family Potyviridae. When examined under a microscope it can be seen to have flexuous filamentous particles 752 nm in length and 13 nm in width (Demski et al., 1984). Being a *Potyvirus* it consists of a single strand positive sense RNA chain, which in the case of PStV consists of 10,062 nucleotides (Brunt et al., 1996).

5.2.2 Life cycle and dispersal

PStV like all plant viruses requires a susceptible host plant for long term survival and replication.

Infected seed provides the primary source of inoculum (Zettler et al., 1993; Xu et al., 1991), and also provides an overwintering mechanism when hosts are unavailable. PStV can also be transmitted by mechanical inoculation (Dinarto and Ilyas 1996) and the transfer of sap between plants (Mishra et al., 1993). PStV can also be spread by several aphid vectors.

PStV is transmitted in a non-persistent, non-circulative manner (Demski et al., 1984) by five aphids; Cowpea aphid (*A. craccivora*) (Sreenivasulu and Demski 1988; Adalla and Natural 1988; Choopanya and Kittipakorn 1989), Cotton aphid (*A. gossypii*) (Adalla and Natural 1988; Choopanya and Kittipakorn 1989), Soybean aphid (*A. glycines*) (Choopanya and Kittipakorn 1989), Rusty plum aphid (*H. setariae*) (Saleh et al., 1989) and Green peach aphid (*M. persicae*) (Sreenivasulu and Demski 1988) all of which are currently present in Australia (see Table 3). Being non-persistent and non-

circulative the virus is only spread by the aphid for a relatively short period of time after the initial acquisition of the virus.

This suggests that the most likely way that this virus could enter Australia is by the importation and subsequent planting of seed containing the virus, rather than the importation of aphids (which only carry the virus for a short time). Should the virus enter Australia endemic aphids, such as the five species listed in Table 3, would be able to spread the virus between plants, paddocks and production areas. Movement restrictions on seed and live host plants, and the removal of hosts (including volunteer plants and non-crop hosts) from the area will limit the spread of the virus.

5.2.3 Host range

The major host of PStV is the peanut or groundnut (*Arachis hypogaea*), however there are other leguminous crops that can act as hosts for this virus. For example soybeans (*Glycine max*) (Zettler et al., 1993), cowpeas *(Vigna unguiculata),* indigo (*Indigofera amoen*)*,* puero (*Pueraria phaseoloides*)*, Stylosanthes capitata* and S. *craba* (Mishra et al., 1993) can all act as hosts of the virus.

It is also possible that other members of the Fabaceae (legume) family could act as hosts for this virus. Further hosts are listed in Section 11.1 Appendix 1, which provides a list of PStV's host species as listed by CABI (2013).

5.2.4 Current geographic distribution

PStV is currently known to occur in Asia, Africa and North America.

In Asia the virus has been reported from: India (Prasada Rao et al., 1991), South Korea (Choi et al., 2001), Vietnam (Mishra et al., 1993), Japan (Senboku 1989), Taiwan (Vetten et al., 1992), China, Indonesia, Thailand, Malaysia and the Philippines (Prasada Rao et al., 1991).

The virus also occurs in parts of the USA (Prasada Rao et al., 1991), South Africa (Higgins et al., 1999) and Senegal (CABI 2013).

This information suggests that peanut seed from these countries could potentially pose a risk of introducing this virus into Australia.

5.2.5 Potential geographic distribution in Australia

PStV has a wide geographic range indicating its ability to adapt and spread to new areas. Numerous hosts of the virus are grown in Australia (see Section 5.2.4) and known insect vectors of PStV are also widespread in Australia. This information suggests that areas of Australia that grow susceptible hosts, such as peanut and soybean growing areas of Australia (e.g. parts of Queensland, Northern NSW) could be affected by this virus.

5.2.6 Symptoms

Symptoms of PStV depend on the strain of the virus and the host species/cultivar. When a plant becomes infected the leaves become discoloured developing dark green coloured stripes along the lateral veins. As the leaf ages more of the leaf becomes discoloured taking on a so called "oak leaf" pattern (which is when a large proportion of the leaf becomes a dark green colour) (Lynch et al.,

1988). The damage to the leaves reduces the productivity of the plant and causes reduced yields and vigour.

5.2.7 Diagnostic information

Currently there is not an endorsed National Diagnostic Protocol for PStV. However the Pest Risk Review by Coutts (2009) suggests that laboratories in Western Australia, Queensland and Victoria would have the capability to identify PStV using Electron microscopes, ELISA or RT-PCR techniques.

Section 11.3 Appendix 3 provides further information on diagnostic facilities and advisory services that can be utilised in the event of an incursion.

ELECTRON MICROSCOPY

Electron microscopy can be used to examine sap extracts for characteristic particle sizes and shapes. The discovery of particular particle shapes and sizes (i.e. the presence of flexuous filamentous particles 752 nm in length and 13 nm in width (Demski *et al*., 1984)) would help confirm the presence of the virus.

Diagnosis using electron microscopy requires validation by ELISA and/or RT-PCR.

ELISA

Sreenivasulu and Demski (1988) used Enzyme Linked Immunosorbent Assay (ELISA) in their PStV inoculation experiment and were able to identify the virus, in the leaves of the inoculated plants three weeks after the peanut plants were inoculated.

Xu et al., (1991) used Direct Antigen-Coated, indirect ELISA (ELISA - DAC) to detect PStV in peanut seeds. Similarly, Pinnow et al., (1990) also used ELISA to identify the virus in peanut seeds.

This means that both leaf and seeds are able to be tested for the presence of PStV using ELISA tests.

RT-PCR

The Reverse Transcription Polymerase Chain Reaction (RT-PCR) is a rapid, specific, and sensitive test that can be used to detect and diagnose PStV. For example Gillaspie et al., (2000) developed an Immunocapture-RT-PCR (IC-RT-PCR) technique to test peanut seeds for PStV.

RT-PCR is very specific and sensitive and allows for the detection of minimal amounts of target RNA. As such, RT-PCR can be used to validate the results from electron microscopy and ELISA.

5.2.8 Pest risk analysis – PStV

Potential or impact	Rating
Entry potential	Medium
Establishment potential	High
Spread potential	High
Economic impact	High
Overall risk	High

ENTRY POTENTIAL

Rating: Medium

PStV occurs in a number of peanut producing countries in Asia, Africa and North America (see Section 5.2.4). PStV can be spread by the importation and planting of infected peanut seed (Zettler et al., 1993). Currently there are restrictions in place to reduce this risk; however such protocols cannot control illegal importation of seeds or plants. There is also a minor risk of virus carrying insects arriving and carrying the disease into Australia, although this is a very low risk given the fact that the virus does not persist for long periods in insect vectors.

Based on this information the entry potential of this virus is considered to be **Medium**.

ESTABLISHMENT POTENTIAL

Rating: High

Peanuts and a number of other host plants are widely planted in Australia. Australia also has all five of the aphid vectors identified as vectoring the virus. This means that should the virus be introduced to Australia it could rapidly become established due to the presence of local vector populations and host plants.

This suggests that the establishment potential of PStV is likely to be **High**.

SPREAD POTENTIAL

Rating: High

As PStV is known to be seed-borne (Zettler et al., 1993) and the known vectors of this virus all occur in Australia the spread potential of PStV can be considered as being **High**.

ECONOMIC IMPACT

Rating: High

PStV has caused significant yield losses in infected peanut crops in Indonesia, where losses of 30 - 40% have been recorded (Saleh et al., 1992). Soybeans can also suffer yield losses (Zettler et al., 1993).

Therefore the economic impact of PStV in Australia is likely to be **High.**

OVERALL RISK

Rating: High

Based on the individual ratings above, the combined overall risk of PStV is considered to be **High**.

5.3 Pest Details – Example 2: Chickpea Chlorotic Dwarf Virus

Common name:	Chickpea Chlorotic Dwarf Virus (CpCDV),
Scientific name:	*Chickpea Chlorotic Dwarf Virus* (*Geminivirus*)
Taxonomic position:	Group: Group II (ssDNA virus) Family: Geminiviridae Genus: *Geminivirus*

5.3.1 Background

Chickpea chlorotic dwarf virus (CpCDV) causes significant yield losses on infected chickpea crops overseas. For example Horn et al., (1996) suggest losses of 75-100% in chickpea crops in India and Pakistan. With higher losses occurring if the infection occurs prior to flowering rather than during flowering (Akhtar et al., 2011)

CpCDV is a member of the genus *Geminivirus* and the family Geminiviridae meaning that it consists of a single strand of DNA. CpCDV consists of 2,900 nucleotides in a circular arrangement (Horn et al., 1993).

5.3.2 Life cycle and dispersal

Leafhopper vectors spread the virus while feeding. This virus is only known to be dispersed by *Orosius orientalis* (Farzadfar et al., 2008, Horn et al., 1994), *O. albicinctus* (Akhtar et al., 2011) and *Neolimnus aegyptiacus* (Hamed and Makkouk 2002).

Horn et al., (1994) found that *O. orientalis* could acquire the virus in less than 2 minutes and begin infecting plants in as little as 2 hours. They also found that *O. orientalis* could retain the virus for up to 21 days and that the insect didn't lose the virus if it moulted after acquiring the virus. This suggests that CpCDV is held in the vector in a persistent manner. *Geminivirus* viruses, including CpCDV, are vectored in a persistent, circulative and non-propagative manner (Abraham et al., 2010).

There is no reference to this virus being seed-borne, so the spread of seed is unlikely to disperse this virus. Instead overwintering of this virus would appear to depend on alternative host plants or survival in leafhopper vectors. CpCDV was found to not be mechanically transmitted or transmitted by grafting in sugar beet (*Beta vulgaris*) plants in Iran (Farzadfar et al., 2008).

This would suggest that the spread of CpCDV can be managed by controlling the spread of the Cicadellidae vectors and the movement of live host plants.

5.3.3 Host range

Chickpea Chlorotic Dwarf Virus is a major pathogen of chickpeas, however the virus also affects other leguminous and non-leguminous crops. A list of the main host plants is given in Table 4.

Table 4. *Recorded hosts of CpCDV*

Scientific name	Common name	Family	Reference
Beta vulgaris	Sugar beet	Amaranthaceae	Farzadfar et al., (2002); Farzadfar et al., (2008).
Cicer arietinum	Chickpea	Fabaceae	Makkouk et al., (2002); Makkouk et al., (1995)
Lens culinaris	Lentils	Fabaceae	Makkouk et al., (2002)
Phaseolus vulgaris	Common (navy) bean	Fabaceae	Farzadfar et al., (2002)
Pisum sativum	Field pea	Fabaceae	Varma and Malathi (2003)
Vicia faba	Faba bean	Fabaceae	Makkouk et al., (1995)

5.3.4 Current geographic distribution

CpCDV is found in many chickpea producing countries in northern Africa and southern Asia. CpCDV is found in Syria (Kumari et al., 2004), India (Horn et al., 1993), Pakistan (Akhtar et al., 2011), Iran, Iraq, Yemen, Egypt, Sudan (Kumari et al., 2006), Ethiopia (Abraham et al., 2000) and Eritrea (Kumari et al., 2008).

5.3.5 Potential geographic distribution in Australia

This virus currently occurs in northern Africa and parts of southern Asia. Numerous hosts can be affected and many are commercially grown in Australia. The Common brown leafhopper (*Orosius orientalis*), which vectors the pathogen, occurs in Australia. This information suggests that areas of Australia that have both suitable hosts and the vector could be affected by the virus.

5.3.6 Symptoms

Symptoms caused by this virus on chickpeas include plant stunting, a shortening of the distances between stem nodes, browning of the phloem and leaf colour changes (Kumari et al., 2006). The leaves of Desi type chickpeas become red while those of Kabuli type chickpeas become yellow when infected by CpCDV (Horn et al., 1993; Kumari et al., 2006).

5.3.7 Diagnostic information

No National Diagnostic Protocol has been developed or endorsed for CpCDV. However electron microscopy, ELISA and PCR can be used to identify this virus in plant samples.

Section 11.3 Appendix 3 provides further information on diagnostic facilities and advisory services that can be utilised in the event of an incursion.

ELECTRON MICROSCOPY

Electron microscopy can be used to examine sap extracts for characteristic particle sizes and shapes. The discovery of particular particle shapes and sizes (for example, the presence of particles approximately 25 nm in diameter is typical of Geminiviridae viruses (Farzadfar et al., 2008)) would help confirm the presence of the virus.

Diagnosis using electron microscopy requires validation by ELISA and/or PCR.

ELISA

Double-antibody sandwich enzyme-linked immunosorbent assay (DAS-ELISA) was used successfully by Kumari et al., (2006) to detect CpCDV in sap dilutions of 1/640. Dot-blot ELISA was also used successfully with sap dilutions of 1/640 and by direct antigen-coating enzyme-linked immunosorbent assay (DAC-ELISA) at dilutions of 1/1280.

PCR

The Polymerase Chain Reaction (PCR) is a rapid, specific, and sensitive test that can be used to detect and diagnose CpCDV. Farzadfar et al., (2008) describes a set of primers used in PCR assays to identify the virus in symptomatic plants.

PCR can be used to validate the results from ELISA.

5.3.8 Pest risk analysis – CpCDV

Potential or impact	Rating
Entry potential	Low
Establishment potential	High
Spread potential	High
Economic impact	High
Overall risk	Medium

ENTRY POTENTIAL

Rating: Low

CpCDV is not listed as being seed-borne, meaning that that this virus would most likely enter Australia through either an infected leafhopper, or the importation of an infected live host plant. *O. orientalis*, one of the vectors of CpCDV, is able to retain the virus for up to 21 days (Horn et al., 1994), which means that it may be possible for an individual to acquire the virus, enter Australia and find a susceptible host, however the risk of this combination of events happening is likely to be low. There are also quarantine protocols in place to minimise the risk of importing pests and diseases through live plant material.

Therefore the entry potential of this virus into Australia is considered to be **Low**.

ESTABLISHMENT POTENTIAL

Rating: High

One of the three vectors, Common brown leafhopper (*O. orientalis*) (which is also the main vector recorded in the literature), is widespread in Australia. CpCDV host plants (see: Table 4) are widely grown throughout Australia as grain crops.

The presence of an endemic vector and suitable host plants means that the establishment potential of this virus is **High**.

SPREAD POTENTIAL

Rating: High

Due to the large areas planted to susceptible host plants in Australia and the widespread presence of *O. orientalis*, a known vector of the virus, the potential for spread of CpCDV following establishment is considered to be **High**.

ECONOMIC IMPACT

Rating: High

CpCDV causes yield losses on infected chickpea crops overseas. With losses of 75 – 100 % recorded from chickpea crops in India and Pakistan (Horn et al., 1996). This suggests that CpCDV has the capability to cause significant yield losses and have a significant economic effect on chickpea growers. Other leguminous crops are also affected, e.g. faba beans (Makkouk et al., 1995), suggesting that not only chickpeas would be affected if the virus was to establish in Australia.

Based on this information the economic impact of this virus entering Australia is expected to be **High**.

OVERALL RISK

Rating: Medium

Based on the individual ratings above, the combined overall risk is considered to be **Medium**.

5.4 Pest Details – Example 3: Chickpea Chlorotic Stunt Virus

Common name:	Chickpea Chlorotic Stunt Virus (CpCSV)
Scientific name:	*Chickpea Chlorotic Stunt Virus* (*Polerovirus*)
Taxonomic position:	Group: Group IV (Positive sense ssRNA virus) Family: Luteoviridae Genus: *Polerovirus*

5.4.1 Background

Chickpea chlorotic stunt virus (CpCSV) is considered to pose a significant impact on the grains industry and is able to cause significant yield losses on infected chickpea, faba bean and other leguminous crops overseas.

CpCSV is a member of the genus *Polerovirus* and the family Luteoviridae meaning that it consists of a single strand of positive sense RNA. The CpCSV genome consists of 5,900 nucleotides. The viral particles are smooth and approximately 28 nm in diameter (Abraham et al., 2006). This virus is known to be transmitted by aphids which occur in Australia (see Table 3), and is vectored in a persistent, circulative and non-propagative manner. Like other Luteoviridae viruses, CpCSV is not known to be mechanically transmitted.

5.4.2 Life cycle and dispersal

Aphid vectors spread the virus between infected and non-infected plants while feeding. Abraham et al., (2006) suggests that the aphids transmit the virus in a persistent circulative manner, meaning the aphids can carry inoculum for long periods of time. Weeds and volunteer plants can also act as reservoirs of the virus allowing the virus to survive in the absence of vectors and crop hosts.

CpCSV is known to be transmitted by the Cowpea aphid (*A. craccivora*) and the Pea aphid (*A. pisum*) (Asaad et al., 2009; see Table 2). Luteoviridae viruses are not usually seed transmitted (Abraham et al., 2009) and there is no reference to seed transmission of this virus in the literature. Therefore, the virus could be transported on live plants or by the movement of insect vectors.

5.4.3 Host range

Most of the hosts of CpCSV that have been identified are members of the Fabaceae family of plants. However there are some exceptions including plants in the Apiaceae, Brassicaceae, Euphorbiaceae and Solanaceae families. Table 5 lists the hosts of CpCSV.

In the event of an incursion all potential host plants should be considered in any survey and containment programs.

Table 5. *Main hosts of CpCSV*

Scientific name	Common name	Family	Reference
Apium spp.	Celery, etc.	Apiaceae	Asaad et al., (2009)
Cicer arietinum	Chickpea	Fabaceae	Asaad et al., (2009)
Euphorbia spp.	Euphorbias	Euphorbiaceae	Asaad et al., (2009)
Lens culinaris	Lentil	Fabaceae	Asaad et al., (2009)
Medicago spp.	Medics, lucerne	Fabaceae	Asaad et al., (2009)
Physalis longifolia	Common Groundcherry	Solanaceae	Asaad et al., (2009)
Pisum sativum	Field pea	Fabaceae	Asaad et al., (2009)
Sinapis arvensis	Field mustard	Brassicaceae	Asaad et al., (2009)
Trigonella foenum-graecum	Fenugreek	Fabaceae	Abraham et al., (2009)
Vicia ervilia	Bitter vetch	Fabaceae	Asaad et al., (2009)
Vicia faba	Faba bean	Fabaceae	Asaad et al., (2009)
Vicia narbonensis	Narbonne vetch	Fabaceae	Asaad et al., (2009)
Vicia sativa	Common vetch	Fabaceae	Asaad et al., (2009)

5.4.4 Current geographic distribution

CpCSV is found in countries in eastern and northern Africa and parts of western Asia. A list of countries where the virus is known to occur is provided in Table 6.

Table 6 *Geographic distribution of CpCSV*

Country	Reference
Azerbaijan	Mustafayev et al., 2011)
Egypt	Abraham et al., (2009)
Eritrea	Kumari et al., (2008)
Ethiopia	Abraham et al., (2009)
Iran	Bananej et al., (2010)
Morocco	Abraham et al., (2009)
Sudan	Abraham et al., (2009)
Syria	Abraham et al., (2009)
Tunisia	Najar et al., (2011)

5.4.5 Potential geographic distribution in Australia

Parts of Australia have similar climates to affected countries in northern Africa and western Asia (see Section 5.4.4 above). A number of CpCSV hosts are commercially grown in Australia as pulse and fodder crops. Both aphid vectors currently occur in Australia. This information suggests that there is a potential for large areas of the Australian grain belt to be affected by this virus if an incursion was to occur.

5.4.6 Symptoms

The virus causes the leaves of infected plants to yellow and the plants to become stunted (Abraham et al., 2006). Significant yield losses can also be associated with a CpCSV infection.

The severity of symptoms varies depending on the origin of the virus. Abraham et al., (2009) found Syrian isolates of the virus to be more damaging (i.e. cause greater stunting and more yellowing of the leaves) than Ethiopian isolates on the faba bean cultivar “Condor”.

5.4.7 Diagnostic information

No National Diagnostic Protocol has been developed or endorsed for CpCSV. However electron microscopy, ELISA and RT-PCR can be used to identify this virus in plant samples.

Section 11.3 Appendix 3 provides further information on diagnostic facilities and advisory services that can be utilised in the event of an incursion.

ELECTRON MICROSCOPY

Electron microscopy can be used to examine sap extracts for characteristic particle sizes and shapes. The discovery of particular particle shapes and sizes (for example, the presence of particles ~28 nm in diameter and slightly hexagonal in shape is typical of CpCSV (Abraham et al., 2006)) can aid in the diagnosis of the virus.

Diagnosis using electron microscopy requires validation by ELISA and/or RT-PCR.

ELISA

CpCSV can be detected using enzyme-linked immunosorbent assay (ELISA) techniques. For example Abraham et al., (2006) used double antibody sandwich ELISA and triple antibody sandwich ELISA to identify CpCSV affecting legumes in Ethiopia. Both of these tests were also applied by Abraham et al., (2009) to identify CpCSV in faba bean and chickpea.

RT-PCR

The Reverse Transcription Polymerase Chain Reaction (RT-PCR) is a rapid, specific, and sensitive test that can be used to detect and diagnose CpCSV from extracted nucleic acids. RT-PCR has been used by Abraham et al., (2009) to identify CpCSV in chickpea and faba bean samples.

A generic RT-PCR method has also been developed by Chomic et al., (2010) to detect Luteoviridae viruses.

5.4.8 Pest risk analysis – CpCSV

Potential or impact	Rating
Entry potential	Low
Establishment potential	High
Spread potential	High
Economic impact	High
Overall risk	Medium

ENTRY POTENTIAL

Rating: Low

CpCSV is currently found in northern Africa and western Asia (see Section 5.4.4). The virus is not known to be seed-borne. Therefore the virus could enter Australia by the entry of an infected insect vector (virus is carried by the vectors in a persistent circulative manner (Abraham et al., 2006)) or the importation of an infected live host plant. The probability of either scenario occurring is likely to be low.

Therefore the entry potential of CpCSV into Australia is considered to be **Low**.

ESTABLISHMENT POTENTIAL

Rating: High

Hosts of CpCSV (Table 6) are widely grown in Australia as crops. Both aphid vectors of the virus are also present in Australia (Table 3).

The presence of both aphid vectors and numerous host plants means that the establishment potential of this virus in Australia is **High**.

SPREAD POTENTIAL

Rating: High

The presence of both vectors (Table 3) and suitable hosts plants (Table 6) in Australia means that the potential for spread of CpCSV following establishment is considered to be **High**.

ECONOMIC IMPACT

Rating: High

CpCSV can cause significant yield losses on infected crops (which include chickpea, lentil, field pea, faba bean and vetch). This suggests that CpCSV has the capability to have a significant economic effect on grain legume producers. Some legumes grown for fodder are also reported as being infected by CpCSV, such as *Medicago* spp. and vetches (*Vicia* spp.) (Asaad et al., 2009).

Based on this information the economic impact of this virus entering Australia is expected to be **High**.

OVERALL RISK

Rating: Medium

Based on the individual ratings above, the combined overall risk is considered to be **Medium**.

6 Pest information/status – sap-sucking insect vectors

6.1 General information on vectors

Table 7 provides a summary of the classification and synonyms of the 7 endemic and 2 exotic vectors of the three example viruses used in this generic contingency plan.

Table 7. *Examples of aphid vectors of exotic virus threats to the Grain Industry*

Scientific name	Synonyms	Common name	Taxonomic position
Acyrthosiphon pisum Harris, 1776	*Acyrthosiphon destructor*; *Acyrthosiphon nigricantis; Acyrthosiphon onobrychis*; *Acyrthosiphon pisi*; *Acyrthosiphon pisum*; *Acyrthosiphon spartii*; *Acyrthosiphon spartii nigricantis*; *Acyrthosiphon trifolii, Anuraphis promedicaginis*; *Aphis basalis*; *Aphis lathyri*; *Aphis onobrychis*; *Aphis onobrychis galegae*; *Aphis pisi*; *Aphis pisi*; *Aphis pisi brevicaudatum*; *Aphis pisi turanicum*; *Aphis pisi ussuriense*; *Aphis pisum*; *Macchiatiella promedicaginis*; *Macchiatiella trifolii; Macrosiphon theobaldii*; *Macrosiphum destructor*; *Macrosiphum onobrychis; Macrosiphum pisi*; *Macrosiphum pisum*; *Macrosiphum spartii*; *Macrosiphum trifolii; Macrosiphum trifolii; Nectarophora destructor*; *Siphonophora corydalis*; *Siphonophora ononis*; *Siphonophora ononis*; *Siphonophora pisum ononis*; *Siphonophora spartii*; *Siphonophora spartii nigricantis*	Pea aphid	Kingdom: Amimalia Phylum: Arthropoda Subphylum: Uniramia Class: Insecta Order: Hemiptera Superfamily: Aphidoidea Family: Aphididae
Aphis craccivora Koch, 1854	*Aphis atronitens*; *Aphis beccarii*; *Aphis cistiella*; *Aphis citricola*; *Aphis dolichi*; *Aphis hordei*; *Aphis isabellina*; *Aphis kyberi*; *Aphis laburni*; *Aphis leguminosae*; *Aphis loti*; *Aphis medicaginis*; *Aphis mimosae*; Aphis oxalina; *Aphis papilionacearum*; *Aphis robiniae*; *Doralida loti*; *Doralina craccivora*; *Doralina medicaginis*; *Doralina salsolae*; *Doralis laburni*; *Doralis medicaginis*; *Doralis meliloti*; *Doralis robiniae*; *Pergandeida craccivora*; *Pergandeida loti*; *Pergandeida medicaginis*; *Pergandeida robiniae*	Cowpea aphid	Kingdom: Amimalia Phylum: Arthropoda Subphylum: Uniramia Class: Insecta Order: Hemiptera Superfamily: Aphidoidea Family: Aphididae
Aphis glycines Matsumura, 1917	*Aphis justiceae*	Soybean aphid	Kingdom: Amimalia Phylum: Arthropoda Subphylum: Uniramia Class: Insecta Order: Hemiptera Superfamily: Aphidoidea Family: Aphididae

Scientific name	Synonyms	Common name	Taxonomic position
Aphis gossypii Glover, 1877	*Aphis bauhiniae*; *Aphis circezandis*; *Aphis citri*; *Aphis citrulli*; *Aphis cucumeris*; *Aphis cucurbiti*; *Aphis lilicola*; *Aphis minuta*; *Aphis monardae*; *Aphis parvus*; *Aphis tectonae*; *Cerosipha gossypii*; *Doralina frangulae*; *Doralina gossypii*; *Doralis frangulae*; *Toxoptera leonuri*	Cotton aphid, melon aphid	Kingdom: Amimalia Phylum: Arthropoda Subphylum: Uniramia Class: Insecta Order: Hemiptera Superfamily: Aphidoidea Family: Aphididae
Hysteroneura setariae (Thomas)	*Aphis bituberculata; Aphis prunicolens; Aphis scotti; Aphis setariae; Carolinaia bituberculata; Carolinaia setariae; Carolinia setariae*; *Heteroneura setariae; Hysteroneura bituberculata; Hysteroneura oglobini*	Rusty plum aphid	Kingdom: Amimalia Phylum: Arthropoda Subphylum: Uniramia Class: Insecta Order: Hemiptera Superfamily: Aphidoidea Family: Aphididae
Myzus persicae Sulzer, 1776	*Aphis convolvuli*; *Aphis cynoglossi*; *Aphis derelicta*; *Aphis dianthi*; *Aphis dubia*; *Aphis egressa*; *Aphis malvae*; *Aphis persicae*; *Aphis persiciphila*, *Aphis persola*; *Aphis rapae*; *Aphis redundans*; *Aphis suffragans*; *Aphis tuberoscellae*; *Aphis vastator*; *Aphis vulgaris*; *Aulacorthum convolvuli*; *Myzodes persicae*; *Myzodes tabaci*; *Myzoides persicae*; *Myzus dianthi*; *Myzus malvae*; *Myzus nicotianae*; *Myzus pergandei*; *Myzus persicae* var. *cerastii*; *Myzus persicae* var. *sanguisorbella*; *Nectarosiphon persicae*; *Phorodon cynoglossi*; *Phorodon persicae*; *Rhopalosiphum betae*; *Rhopalosiphum calthae*; *Rhopalosiphum dianthi*; *Rhopalosiphum lactucellum*; *Rhopalosiphum lactucellum*; *Rhopalosiphum persicae*; *Rhopalosiphum solani*; *Rhopalosiphum tuberosellae*; *Rhopalosiphum tulipae*; *Siphonophora achyrantes*; *Siphonophora nasturtii*	Green peach aphid	Kingdom: Amimalia Phylum: Arthropoda Subphylum: Uniramia Class: Insecta Order: Hemiptera Superfamily: Aphidoidea Family: Aphididae
Neolimnus aegyptiacus Matsumura	*Scaphoideus aegyptiacus*	Leafhopper	Kingdom: Amimalia Phylum: Arthropoda Subphylum: Uniramia Class: Insecta Order: Hemiptera Superfamily: Cicadelloidea Family: Cicadellidae

Scientific name	Synonyms	Common name	Taxonomic position
Orosius albicinctus Distant	None	Leafhopper	Kingdom: Amimalia Phylum: Arthropoda Subphylum: Uniramia Class: Insecta Order: Hemiptera Superfamily: Cicadelloidea Family: Cicadellidae
Orosius orientalis Matsumara	*Eutettix orientalis*; *Nesophrosyne orientalis*; *Orosius albicinctus*; *Thamnotettix filigranua.*	Sesame jassid	Kingdom: Amimalia Phylum: Arthropoda Subphylum: Uniramia Class: Insecta Order: Hemiptera Superfamily: Cicadelloidea Family: Cicadellidae

6.1.1 Background

Apart from spreading viruses many of these sap-sucking insects are pests in their own right and can cause damage to various crops. Damage is caused by the insects feeding on host plants. Infested plants often become discoloured at the feeding sites due to chemicals in the insect's saliva. Aphids and leafhoppers also produce honeydew, as a by-product of their feeding process, which encourages sooty mould to develop on the leaf surfaces of infected plants.

Neolimnus aegyptiacus and *Orosius albicinctus* are not present in Australia (see Table 8 for risk ratings). *Acyrthosiphon pisum*, *Aphis craccivora*, *A. gossypii*, *Hysteroneura setariae, Myzus persicae* and *Orosius orientalis* are widely distributed in Australia, while *A. glycines* is restricted to northern NSW and southern Queensland.

Table 8 *Risk ratings of exotic insect vectors (PHA 2009-review 2014)*

Scientific name	Common name	Entry potential	Establishment potential	Spread potential	Economic impact	Overall risk
Neolimnus aegyptiacus	-	MEDIUM	MEDIUM	MEDIUM	LOW	VERY LOW
Orosius albicinctus	-	MEDIUM	MEDIUM	MEDIUM	LOW	VERY LOW

6.1.2 Life cycles

APHIDIDAE

Aphids have a complex lifecycle. Many aphids are able to reproduce via parthenogenesis, that is, females do not need to mate to produce young. In which case no eggs are laid, instead live-nymphs are produced asexually. The aphid lifecycle can be either holocyclic (i.e. complete with both sexual and asexual reproduction) or anholocyclic (i.e. reproduction occurs solely via parthenogenesis with no eggs laid, in which case there are usually no males present in the population).

The time it takes for the development of most aphids is determined by temperature. The reproductive rate of adults is determined by temperature and adult morphology (i.e. is the adult apterous or alate). For example the development of *M. persicae* can be rapid, often 10 to 12 days for a complete generation under optimal conditions, while at cooler temperatures the life cycle may last up to 50 days (Toba 1964). Wingless (apterous) adults were found to lay more eggs than winged (alate) adults. This trend is also true of other Aphididae species.

Specific information is given in Table 9 on the reproduction of each of the Aphididae insect vectors of the three viruses used as examples in this contingency plan.

Table 9 *Species specific information on the reproduction and development of Aphididae vectors*

Scientific name	Notes on development and reproduction
Acyrthosiphon pisum	*Acyrthosiphon pisum* reproduces both via parthenogenesis (young born as nymphs) and sexually (eggs laid, i.e. holocyclic reproduction) (Miura et al., 2003). Morgan et al., (2001) found that the host as well as temperature has an effect on the generational times of this insect. At 26.7 °C it took an average of 8.5 - 8.8 days from birth to adulthood. At 11.9 °C a generation was completed in 16.2 - 16.8 days depending on the cultivar of field pea that the aphids were feeding on.
Aphis craccivora	*Aphis craccivora* is known to reproduce parthenogenetically, especially in warmer climates (Elliot and McDonald 1976). Elliott and McDonald (1976) found that the aphid was able to produce either live young or eggs and that alate adults produced fewer eggs than apterous adults. They also found that apterous adults began reproducing within hours of their final moult whereas alate adults did not reproduce until 1 to 2 days after their final moult. Oguya (1997) also studied this aphid and suggests that *A. craccivora* takes 3 to 5 days to develop from larva to adult under optimal conditions.
Aphis glycines	*Aphis glycines* is known to reproduce parthenogenetically (Zhang et al., 2008) McCornack et al., (2004) studied *A. glycines* reproduction at 20, 25, 30 and 35 °C and found that reproduction was greatest at 25 °C with aphid populations doubling in 1.5 days. Aphids took 4.9±0.1 days to begin reproducing and had a reproductive period of 9.5 days when raised at 25°C.
Aphis gossypii	*Aphis gossypii* is only known to reproduce parthenogenetically (Fuller et al., 1999). Xia et al., (1999) studied this aphid on cotton and assessed its reproduction at 10, 15, 20, 25, 30 and 35°C and found that development was fastest at 30°C. But reproduction was greatest at 25°C. Van Steenis and El-Khawass (1995) found that aphids reached maturity on cucumbers in 4.8 days at 20°C to 3.2 days at 30°C.

Hysteroneura setariae	*Hysteroneura setariae* is known to reproduce parthenogenetically (Carver 1977) Lifecycle consists of 4 instars followed by the adult insect. Each instar takes between 2 and 3 days to develop with the development from the first instar to adult taking approximately 10 days. The insect lives as an adult for 8 - 15 days (Jahn et al., 2006).
Myzus persicae	*M. persicae* reproduce solely by parthenogenesis in warmer climates but reproduce holocyclicly in cooler climates (Mau and Kessing 1991). As a consequence of this, populations in warm areas are composed solely of females. In colder climates, aphids overwinter as eggs and both sexes may be present. Toba (1964) studied this species and found that it has four instars and takes 10 to 12 days for a complete generation in ideal conditions; however in cooler temperatures the life cycle may last up to 50 days.

CICADELLIDAE

Most leafhoppers (family Cicadellidae) reproduce sexually, although some from high altitudes reproduce parthenogenetically (Freytag and Sharkey 2002).

Bindra and Singh (1970) studied *O. albicinctus*, in India and found that the leafhoppers reproduce more slowly at low temperatures. *O. albicinctus* takes between 6.2 days and 95.8 days to hatch depending on the temperature the egg was exposed to. It then takes between 11.6 and 105.3 days for the insect to complete its five nymphal instar phases when exposed to mean temperatures of 34.9°C and 10.8°C respectively.

O. orientalis has a life cycle consisting of an egg followed by five instars. Eggs take between 7 and 22 days to hatch and there can be up to three generations produced each season in both laboratory and field conditions (Trebicki 2010).

No specific information on the lifecycle of *Neolimnus aegyptiacus* was found in a search of the scientific literature.

6.1.3 Dispersal

All of the above Aphididae and Cicadellidae species can potentially be transported long distances by wind and storms, or as hitchhikers on plant material, machinery and equipment.

In the case of the aphids there are both alate and apterous adults in the population. Winged adults are capable of flight and are able to travel further than their wingless counterparts. For example Zhang et al., (2008) looked at the flight capability of *A. glycines* and found that 12 - 24 hour old alate adults were able to fly 4.6 - 5.1 km when conditions were between 16°C and 28°C.

The Cicadellidae species *Neolimnus aegyptiacus, Orosius albicinctus* and *O. orientalis* all have wings as adults. Adult leafhoppers (such as O. orientalis) are capable of short jumps and flights (Trebicki et al., 2010a).

6.2 Affected hosts

6.2.1 Host range

Many aphids and leafhoppers have wide host ranges. Table 10 summarises the hosts that each of the nine vectors are most often associated with.

Table 10 *Selected plant hosts for sap-sucking insect vectors*

Vector	Main host plants	Reference(s)
Pea aphid (*Acyrthosiphon pisum*)	Grass pea (*Lathyrus sativus*)	Wale et al., (2000)
	Lentil (*Lens culinaris*)	Wale et al., (2000)
	Lucerne (*Medicago sativa*)	Cuperus et al., (1982)
	Field pea (*Pisum sativum*)	Morgan et al., (2001); Wale et al., (2000)
	Red clover (*Trifolium pratense*)	Markkula and Roukka (1970)
	Faba bean (*Vicia faba*)	Sutherland (1969); Wale et al., (2000)
Cowpea aphid (*Aphis craccivora*)	Peanut (*Arachis hypogaea*)	Choopanya and Kittipakorn (1989); Sreenivasulu and Demski (1988)
	Chickpea (*Cicer arietinum*)	Abraham et al., (2006)
	Lentil (*Lens culinaris*)	Hossain et al., (2006)
	Medics and lucerne (*Medicago* spp.)	Van Emden and Harrington (2007)
	Cloves (*Trifolium* spp.)	Van Emden and Harrington (2007)
	Faba bean (*Vicia faba*)	Abraham et al., (2006)
	Vetch (*Vicia* spp.)	Van Emden and Harrington (2007)
	Mung bean (*Vigna radiata*)	Purivirojkul et al., (1978)
	Cowpea (*Vigna unguiculata*)	Ofuya (1997)
Soybean aphid (*Aphis glycines*)	Peanut (*Arachis hypogaea*)	Choopanya and Kittipakorn (1989)
	Soybean (*Glycine max*)	Zhang et al., (2008)
	Common buckthorn (*Rhamnus cathartica*) (which it uses to survive over winter)	Zhang et al., (2008)
Cotton aphid (*Aphis gossypii*)	Peanut (*Arachis hypogaea*)	Choopanya and Kittipakorn (1989)
	Melons and cucumbers (Cucurbitaceae)	Martin et al., (2003)
	Cotton (*Gossypium hirsutum*)	Xia et al., (1999)

Vector	Main host plants	Reference(s)
Rusty plum aphid (*Hysteroneura setariae*)	Barleys (*Hordeum* spp.)	Stoetzel (1987)
	Plum (*Prunus domestica*)	Stoetzel (1987)
	Wild rice (*Oryza minuta*)	Barrion and Litsinger (1991)
	Wild rice (*Oryza officinalis*)	Barrion and Litsinger (1991)
	Rice (*Oryza sativa*)	Jahn et al., (2005)
	Sugar cane (*Saccharum officinarum*)	David and Alexander (1984); Harborne (1988)
	Setaria/foxtail millet (*Setaria* spp.)	Stoetzel (1987)
	Sorghum, Johnston grass (*Sorghum* spp.)	Stoetzel (1987)
	Wheat, durum (*Triticum* spp.)	Stoetzel (1987)
Green peach aphid (*Myzus persicae*)	Peanut (*Arachis hypogaea*)	Sreenivasulu and Demski (1988)
	Sugar beet (*Beta vulgaris*)	Thongmeearkom et al., (1976)
	Chickpea (*Cicer arietinum*)	Akhtar et al., (2011)
	Sorghum (*Sorghum bicolor*)	Thongmeearkom et al., (1976)
	Maize (*Zea mays*)	Thongmeearkom et al., (1976)
Neolimnus aegyptiacus	Chickpea (*Cicer arietinum*)	Hamed and Makkouk KM (2002)
	Lime (*Citrus aurantiifolia*)	Alhudaib et al., (2009)
	Sesame (*Sesamum indicum*)	Kalra (1987)
Orosius albicinctus	Chickpea (*Cicer arietinum*)	Akhtar et al., (2011)
	Sunn hemp (*Crotalaria juncea*)	Bindra and Singh (1970)
	Sesame (*Sesamum indicum*)	Mishra (2004)
Common brown leafhopper (*Orosius orientalis*)	Common amaranth (*Amaranthus retroflexus*)	Trebicki et al., (2010b)
	Cape weed (*Cryptostemma calendulaceae*)	Trebicki et al., (2010b)
	Marshmallow (*Malva parviflora*)	Trebicki et al., (2010b)
	Tobacco (*Nicotiana tabacum*)	Trebicki et al., (2010b)
	Bean (*Phaseolus vulgaris*)	Trebicki et al., (2010b)
	Plantain (*Plantago lanceolata*)	Trebicki et al., (2010b)
	Wild radish (*Raphanus raphanistrum*)	Trebicki et al., (2010b)
	White clover (*Trifolium repens*)	Trebicki et al., (2010b)

6.2.2 Current geographic distribution

Seven of the nine vectors identified (Table 3) are already present in Australia and have a global distribution. *Neolimnus aegyptiacus,* and *Orosius albicinctus* do not currently occur in Australia.

N. aegyptiacus currently occurs in parts of Africa (including South Africa, Sudan, Egypt) and Asia (including Israel, Iraq and India (Linnavuori 1961) and Saudi Arabia (Alhudaib et al., 2009)).

O. albicinctus occurs in India (Bindra and Singh 1970), Pakistan (Akhtar et al., 2011) and parts of the Middle East (Esmailzadeh-Hosseini et al., 2007).

6.2.3 Symptoms

Aphids and leafhoppers cause a number of symptoms on plants beyond simply transmitting plant viruses. For example the saliva that these insects inject into the plant while feeding can cause the plants' leaves to discolour and can, in some cases, cause plant death.

Aphids and leafhoppers also produce honeydew which in turn encourages sooty mould to develop, which can have an effect on the plants productivity.

6.3 Diagnostic information

Accurate identification of aphids and leafhoppers to a species level requires dissection and the microscopic examination of the insect. Various texts and papers have been published with diagrams to assist with identification (e.g. Blackman and Eastop 2000; Ghauri 1966 and Linavuori 1953). The following provides a general overview to assist with the preliminary identification of the insects.

6.3.1 Pea aphid (*Acyrthosiphon pisum*)

The pea aphid, *A. pisum,* is a small grey green to lime green coloured aphid. This species has two long green/brown coloured siphunculi and a long single cauda (tail) (see Figure 3). Nymphs are smaller and are often a lighter colour. Adults are approximately 2.2 - 3 mm long with antennae longer than their body length. Both adults and nymphs live in the same areas of the host plant. There are both alate and apterous adult forms. This species occurs in Australia.

Figure 3 *Apterous A. pisum with nymphs, note siphunculi (arrows). Source: Joseph Berger, Bugwood.org*

6.3.2 Cowpea aphid (*Aphis craccivora*)

These are small (body is 1.4 - 2.2 mm long) grey brown to black coloured aphids with two siphunculi and single cauda (Figure 4). This species mostly feeds on legumes. The Cowpea aphid usually lives in large groups consisting of adults and nymphs. There are both alate and apterous adult forms. This species occurs in Australia.

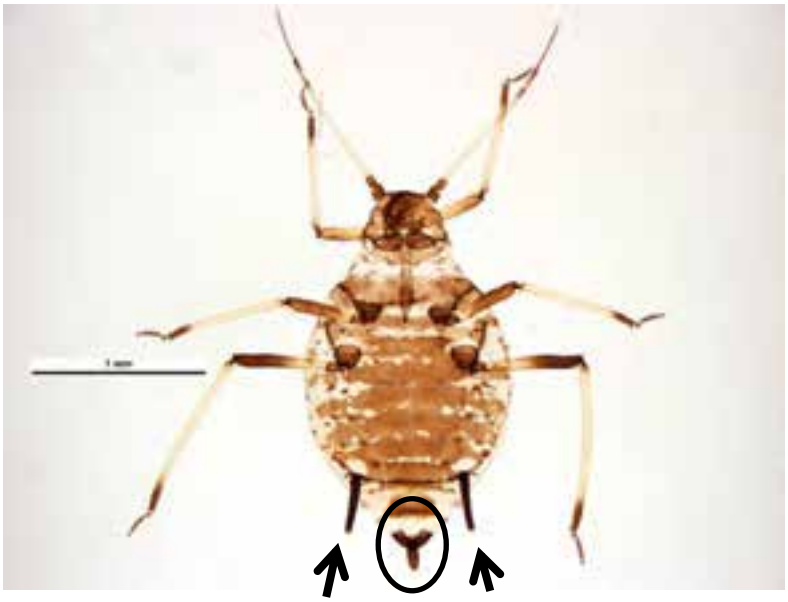

Figure 4 *Close up image of A. craccivora, note siphunculi (arrows) and single cauda (circle). Source: Simon Hinkley & Ken Walker Museum Victoria, PaDIL.*

6.3.3 Soybean aphid (*Aphis glycines*)

Adult Soybean aphids are tiny (~1.3 mm long), green coloured with two darker coloured siphunculi and a single cauda (see Figure 5). Nymphs are usually a yellow-green colour. Both nymphs and adults live in dense clusters on the underside of host plants, such as Soybeans (see Figure 6). This aphid has both alate and apterous adult forms. This species occurs in Australia.

Figure 5 *Apterous adult Soybean aphid, note siphunculi (arrows). Source: Adam Sisson Iowa State University, Bugwood.org*

Figure 6 *A. glycines colony on soybean. Source: Christina Di Fonzo Michigan state university. Bugwood.org*

6.3.4 Cotton aphid (*Aphis gossypii*)

Apterous Cotton aphid adults are approximately 0.9 - 1.8 mm long. There is a large amount of colour variation in this species, aphids can be black, tan, grey, green or white in colour. Alate adults are usually black in colour and are much the same size as the apterous adults with a length of approximately 1.1 - 1.8 mm. The adult possesses two dark siphunculi and a single cauda (Figure 7). These aphids feed on a number of hosts including cotton. This species occurs in Australia.

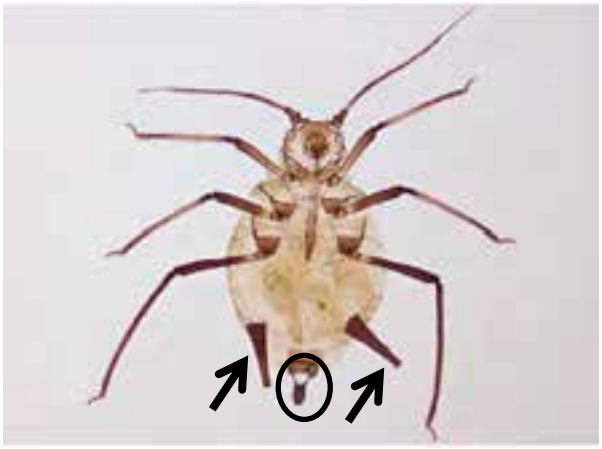

Figure 7 *Apeterous A. gossypii, note siphunculi (arrows) and cauda (circle). Source: Rebecca Graham DAFWA, PaDIL*

6.3.5 Rusty plum aphid (*Hysteroneura setariae*)

The Rusty plum aphid is a 1.5 - 2 mm long, brown coloured aphid (Stoetzel 1987) with two black coloured siphunculi and a single cauda (Figure 8). The wing venation of alate adults can aid in this species identification as the hind wing only has one oblique vein instead of two, which is typical of many other aphids (David and Alexander 1984). This species occurs in Australia.

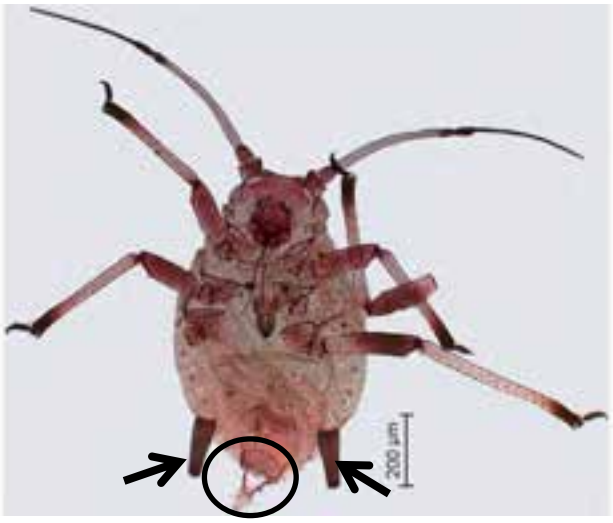

Figure 8 *Apterous H. setariae, note siphunculi (arrows) and cauda (circle). Source: Qing-Hai Fan MAF New Zealand, PaDIL*

6.3.6 Green peach aphid (*Myzus persicae*)

Apterous (wingless) adult Green peach aphids vary greatly in colour. Individuals can be green (Figure 9), pale yellow, pink, red or black. Adults are 1.5 to 2 mm long (Mau and Kessing 1991). Alate (winged) adults have green abdomens with black or dark brown markings, a black thorax and translucent wings (Figure 10).

Nymphs are pale yellow-green in colour with three dark lines on the abdomen. Nymphal development is completed in 6 to 11 days in warmer climates (Toba 1964). In cooler regions, aphids overwinter during the egg stage. The eggs are shiny black and are often laid on the bark of fruit trees. This species occurs in Australia.

Figure 9 *Apterous adult and nymphs. Source: David Cappaert, Michigan State University, Bugwood.org*

Figure 10. *Alate M. persicae. Source: Scott Bauer, USDA Agricultural Research Service, Bugwood.org.*

6.3.7 *Neolimnus aegyptiacus*

N. aegyptiacus is a light brownish coloured leafhopper with red-grey eyes and a typical leafhopper body shape (i.e. similar to *O. orientalis* seen in Figure 11). It is approximately 4.2 mm long. Linnavuori (1953) has further information and diagrams of dissected insects to aid in the identification of this species. This species does not occur in Australia.

6.3.8 *Orosius albicinctus*

O. albicinctus is a small brown coloured leafhopper with irregularly spaced spots on its head and body. Accurate identification requires microscopic examination of the insect's genitalia. The female ranges from 2.6 to 3.3 mm in length, males are slightly smaller with lengths ranging from 2.7 to 2.9 mm. Ghauri (1966) provides more detailed information for the identification of this insect. This species does not occur in Australia.

6.3.9 Common brown leafhopper (*Orosius orientalis*)

Common brown leafhopper (*O. orientalis*) is a common Australian leafhopper. It is a predominantly brown colour with a number of lighter and darker patches giving it a mottled appearance (Figure 11). This species is approximately 3 mm long and slightly over 1 mm wide.

Figure 11 *Adult O. orientalis Source: Piotr Trebicki, Wikimedia commons*

7 Pest management

7.1 Response checklist

The following checklist (Table 11) provides a summary of the response measures to be considered and documented within a Response Plan during an incursion of a new sap-sucking insect transmitted virus into Australia.

Table 11. *Checklist of requirements to be identified in a Response Plan*

Checklist item	Further information
Destruction methods for plant material, soil and disposable items	Sections 8.1.1 and 8.1.2
Disposal issues	Section 8.1.3
Quarantine and movement controls	Section 8.3
Decontamination and hygiene	Section 8.5
Diagnostic information	Sections 5.3.7, 5.3.7, 5.4.7,
Surveillance and tracing	Section 8.6
Surveys and epidemiology	Section 7.2
Zoning	Section 8.4
Communication strategy	Section 11.4

A range of specifically designed procedures for the emergency response to a pest incursion and a general communication strategy refer to PLANTPLAN (Plant Health Australia 2013).

7.2 Surveys and epidemiology studies

Information provided in Sections 7.2.1 to 7.2.3 provides a framework for the development of early detection and delimiting surveys for sap-sucking insect transmitted viruses.

Personnel should avoid moving plant material between production areas to limit movement of both the vector and virus infected plant material. Footwear, tools and vehicle tyres should be thoroughly washed of soil and plant material and then sanitised with a registered disinfectant. Extra precaution should be taken when working in areas known to be infested.

7.2.1 Technical information for planning surveys

When developing surveys for presence and/or distribution of the virus (and potentially for vectors), the following characteristics provide the basic epidemiological knowledge to inform the survey strategy:

- Virus infected plant material may be asymptomatic.
- Host species in Australia are likely to be numerous and widely dispersed and may be present within paddocks, as well as home gardens, landscape plantings and weeds.

- Numerous aphid and leafhopper vectors are already present and widespread in Australia
- Aphid and leafhopper vectors may have hosts that are not the same as the hosts of the virus.
- There is a risk of sap-sucking insect movement on plants, hay, machinery, equipment and personal effects.
- Winged forms of adult aphids and winged leafhoppers can travel long distances on the wind.
- Virus transmission can also occur through mechanical transmission involved with plant management.
- Production areas and significant proportions of Australia have favourable climatic conditions for both virus development and insect vector spread and establishment.

7.2.2 Surveys for early detection of an incursion

Points to consider in effectively monitoring aphid and leafhopper populations are:

- Ensure that the laboratory diagnostician has the relevant diagnostic tools and expertise in the specific virus or insect vector to be identified.
- Initial surveys (using yellow sticky card traps, water pan traps or similar methods) to determine the species of aphid/leafhopper present.
- Sweep nets can also be used to check fields for the presence of aphids, leafhoppers and other sap-sucking insects.
- The position of insect vectors on leaves depends on the insect species and crop. Due of their small size, detection may be dependent on careful visual inspection of plants. The use of a hand lens magnifier may help detect insects.
- If aphids or other sap-sucking insects are detected, leaves infested with insects (preferably with as many life stages as possible) should be collected for identification of the species.

Points to consider in monitoring virus infected material are:

- The host range of the virus must be determined and potential hosts grouped into risk categories in order to trace the transmission and expression of the disease (high, medium and low).
- Conditions under which transmission, amplification and expression of the disease must be determined to assess the likelihood of detection and reporting through general surveillance and to assist with the development of protocols for targeted surveillance.
- Potential pathways for distribution of infected or contaminated material must be determined.
- Depending on the virus, distribution of the virus in the plant may be irregular and plant material with most likely infection should be determined for accurate diagnostics.
- Depending on the virus, host species in Australia may be numerous and widely dispersed and may be present within farms, nurseries, home gardens, landscape plantings or as weeds.
- Virologist expertise will be needed to determine diagnostic protocols and sampling requirements including the age of plant material to be sampled, time of year and the potential to bulk samples from plant species or cultivars.

Important points to consider when developing early detection surveys are:

- Awareness information should be targeted at people who are in regular close contact with potential hosts in high risk areas (e.g. farmers, agronomists).
- Systematic and careful inspection of grain crops is essential to prevent introduction of a sap-sucking insect transmitted virus and limit spread within and from contaminated areas. Early detection of disease symptoms while at low levels, will provide the best chance of eradication.
- Personnel involved in surveys must be trained to recognise particular insect vector(s), the virus symptoms and other similar disorders for comparison.

7.2.3 Delimiting surveys in the event of an incursion

Delimiting surveys should comprise local surveys around the area of initial detection concentrating on areas of reduced or unusual crop growth or where disease symptoms are obvious. Symptomatic plants should be tested to confirm the presence of the virus/vector followed by random sampling from within the same crop to estimate the pest incidence. Surrounding host crops should then be surveyed to determine the extent of the incursion and to inform further survey work.

If the virus can be seed transmitted seed trace-back investigations will indicate how many seed lots and crops will need to be tested. If the seed used has been sown at several sites, delimiting surveys should be conducted at each site.

Delimiting surveys are essential to determine the extent of the incursion and inform the decision-making process. When establishing delimiting surveys the following should be considered:

- The size of the survey area will depend on the size of the infected area and the severity of the infection. Other influencing factors include: distribution pathways for plant material and potentially weather patterns during the period prior to detection (which can influence the spread of insect vectors and therefore the virus) (Figure 12). Other considerations are, the movement of people, plant material or equipment as a result of trace-forward and trace-back investigations.
- Adult leafhoppers and alate aphids can fly and can readily spread long distances by winds or can be transported on infested plants. New introductions can pose serious threats and complicate identification of naturalised populations.
- All potential host species of the virus and/or vector (for specific hosts refer to Sections 5.2.3, 5.3.3 and 5.4.3, (viruses) and Section 6.2.1 (vectors)) should be surveyed, with particular attention paid to the species in which the virus was initially detected.
- In addition to inspection of possible host plants, material should be collected for ongoing diagnostic purposes (refer to Section 7.2.4).
- If the incursion is in a populated area, publication and distribution of information sheets and appeals for public assistance may be helpful.

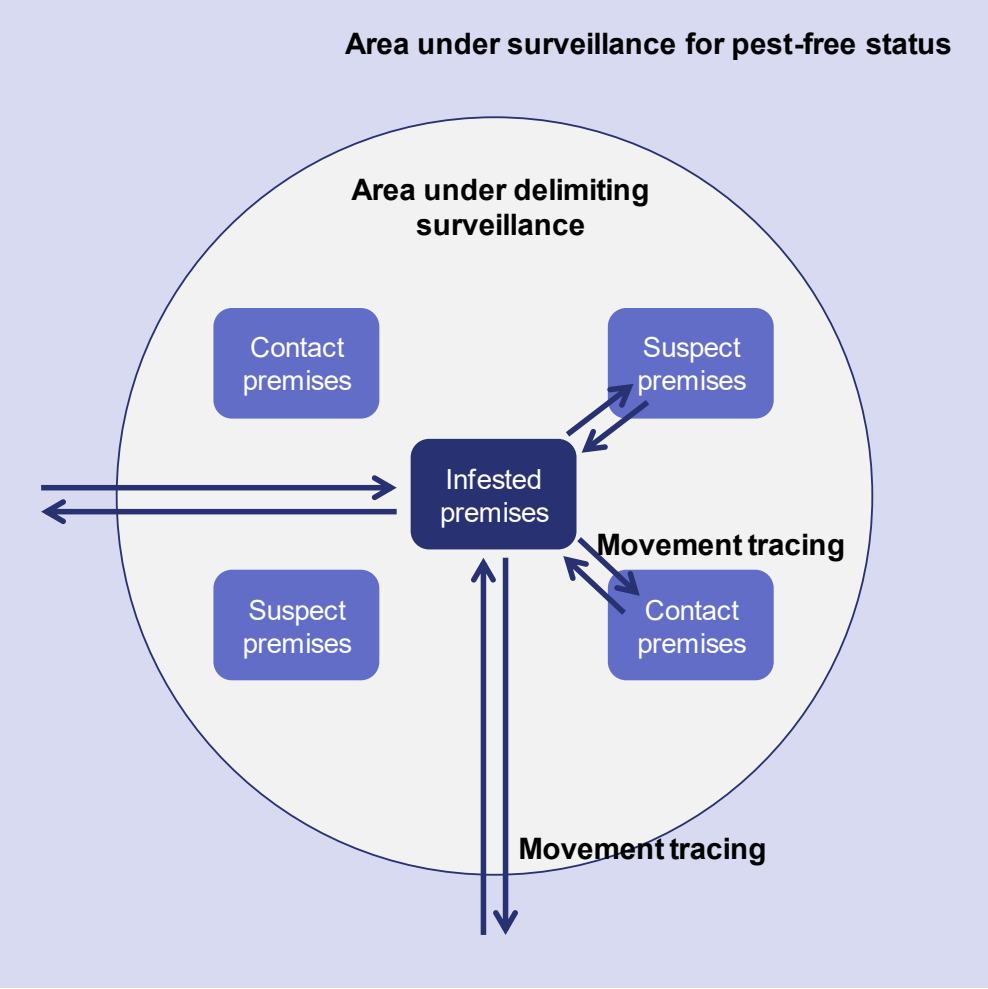

Figure 12. *Diagram of a delimiting survey showing surveillance activities from the infected premises*

7.2.4 Collection and treatment of samples

Once initial samples have been received and preliminary diagnosis made, follow up samples to confirm identification of the virus/vector will be necessary. This will involve sampling directly from the infected crop, and sampling crops over a larger area to determine the extent of the pest's distribution.

At least 100 plants should be taken at random from each site being surveyed. The exact number of samples and survey design will depend on the crop and pest being surveyed for and the statistical confidence required. However, preference may be given to symptomatic plants in fields where the disease incidence is low.

All plants should be assessed for the presence of the pathogen's symptoms. See Sections 5.2.6, 5.3.6 and 5.4.6 for further details on the symptoms caused by the three example viruses.

Protocols for the collection, transport and diagnosis of suspect Emergency Plant Pests (EPPs) must follow PLANTPLAN (Plant Health Australia 2013). Details are provided in the Standard Operating Procedure (SOP) for *Collection and transport of EPPs* available as a supporting document of PLANTPLAN (Plant Health Australia, 2013) (**www.planthealthaustralia.com.au/wp-content/uploads/2013/12/SOP-Collection-and-transport-of-EPPs.pdf**). Any personnel collecting samples for assessment should notify the diagnostic laboratory prior to submitting samples to ensure

expertise is available to undertake the diagnosis.

The total number of samples collected may run into the hundreds or even thousands. It is vital that a system of sample identification is determined early in the procedure to allow for rapid sample processing and accurate recording of results. Samples should be initially collected over a representative area of the infected crop to determine the disease distribution. Plants showing visual symptoms may appear in discrete patches or spread throughout the crop depending on the source of the pathogen.

It is important to note the distribution of disease throughout the crop, as this may indicate whether the pathogens entry and spread was due to an insect vector, or if it was carried on plant material from adjacent paddocks or originated from contaminated machinery or human movement.

It is important that all personnel involved in crop sampling and inspections take all precautions to minimise the risk of disease spread between crops by decontaminating between paddocks.

All sample containers should be clearly labelled with the name, address and contact phone number of both the sending and receiving officers. Containers should be carefully sealed to prevent loss, contamination or tampering of samples. The Chief Plant Health Manager will select the preferred laboratory. Additional labelling includes the identification of affected plant species/parts, the location of the property/paddock (preferably with a GPS reading) as well as symptoms and an image if available. For further information on the appropriate methods to use, refer to the SOP for the Collection and transport of EPPs, available as a supporting document of PLANTPLAN (Plant Health Australia, 2013) (**www.planthealthaustralia.com.au/wp-content/uploads/2013/12/SOP-Collection-and-transport-of-EPPs.pdf**).

COLLECTION AND TREATMENT OF INSECT SPECIMENS

Sampling procedures

Samples of aphids, leafhoppers and other sap-sucking insects can be collected on leaf samples, yellow sticky traps, suction traps or water pan traps.

Both insect and infected plant material samples should be treated in a manner that allows them to arrive at the laboratory in a fresh, well-preserved state. An esky with ice packs or portable fridge should be carried when sampling crops. For appropriate labelling and packaging procedures for suspect EPPs consult the SOP for the Collection and transport of EPPs available as a supporting document of PLANTPLAN (Plant Health Australia, 2013) (**www.planthealthaustralia.com.au/wp-content/uploads/2013/12/SOP-Collection-and-transport-of-EPPs.pdf**).

7.2.5 Collection and treatment of virus samples

In general, plants showing virus like symptoms or suspected symptoms should be sampled.

For CpCDV and CpCSV leaf samples are required (Kumari et al., 2006 and Abraham et al., 2006 respectively). For PStV, samples can be taken from leaf tissue or from the cotyledons of peanut seeds (Gillaspie et al., 2000).

7.2.6 Epidemiological study

There are many factors that affect the development of virus outbreaks in the field. These include: the presence of virulent strains in the environment, susceptibility of the crop varieties, presence of suitable insect vectors and climatic conditions (which can influence the reproduction and spread of the vector).

The number of infected plants within a crop will depend on the source of the virus, presence of suitable vectors and whether environmental conditions have been favourable for the vector to spread the virus from the original infection site.

Sampling of crops within a district and beyond will be based upon the origins of the initial suspect sample(s). Factors to consider will be:

- The proximity of other susceptible plants to the initial infected area(s), including both the current and previous growing seasons. This will include crops on the infected property and those on neighbouring properties. Alternative hosts should also be considered, including weeds, fodder and garden plants.
- Machinery or vehicles that have been into the known infected area or in close proximity to the source of the pathogen or insect vector. This is especially important due to machinery and vehicles possibly moving the virus on adhering plant material or insect vectors.
- The extent of human movements into and around the infected area. A possible link to the recent importation of plant material (including seed if the virus is seed-borne) from other regions should also be considered.
- The source of any seed or live propagation material used on the farm.
- If any other crops have been grown from the same seed source (important if the virus is seed-borne).
- The temperature and environmental conditions. Temperature and environmental conditions will help determine the reproduction and spread of insect vectors.
- The direction of the prevailing wind. The dispersal of insects such as aphids, thrips and leafhoppers can be assisted by the wind.
- Many plant viruses can be spread by mechanical transmission in sap between plants.

7.2.7 Models of spread potential

No models of spread potential have been developed for PStV, CpCDV or CpCSV. If models were to be developed they would need to consider both the virus and vectors ability to spread (which would likely be influenced by climatic conditions) and the availability of suitable host plants.

7.2.8 Pest Free Area guidelines

The establishment and maintenance of Pest Free Areas (PFAs) is a resource-intensive process. Prior to development of a PFA consideration should be given to alternative methods (e.g. treatments or enclosed quarantine) that achieve an equivalent biosecurity outcome to a PFA. A benefit-cost analysis is useful for this purpose.

Determination of PFAs should be completed in accordance with the International Standards for Phytosanitary Measures (ISPMs) 8 and 10 (IPPC 1998a, 1999).

Additional information is provided by the IPPC (1995) in Requirements for the Establishment of Pest Free Areas. This standard describes the requirements for the establishment and use of Pest Free Areas as a risk management option for phytosanitary certification of plants and plant products. Establishment and maintenance of a PFA can vary according to the biology of the pest, pest survival potential, means of dispersal, availability of host plants, restrictions on movement of produce, as well as PFA characteristics (size, degree of isolation and ecological conditions).

In the event of an incursion, specific guidelines for surveys and monitoring will be provided by the Consultative Committee on Emergency Plant Pests (CCEPP). General points to consider are:

- Design of a statistical delimiting survey for symptoms on host plants (see Section 7.2 for points to consider in the design).
- In general plant sampling should be based on at least 100 plants taken at random per crop.
- Seed sampling (if virus is known to be seed-borne) should be based on a minimum of 400 seeds (preferably 1000) as infection levels in seed may be low.
- Preliminary diagnosis can be based on symptoms and morphology.
- PCR, or other molecular methods for confirmation of diagnosis.
- Surveys should also consider alternative hosts of the vector(s) (see Table 10) and the virus (see Sections 5.2.3, 5.3.3 and 5.4.3) and not be limited to the primary infected host.
- Information (including absence of the pest) should be recorded.

7.3 Availability of control methods

7.3.1 Priorities

- Confirm the presence of the pest.
- Limit movement or people and prevent movement of vehicles and equipment through affected areas.
- Stop the movement of any plant material (including seed) that may be infested with the vector or virus.
- Determine the strategy for the eradication/decontamination of the vector, alternative hosts and infested host material.
- Determine the extent of infestation through survey and plant material trace back and trace forward which would be assessed on a case by case basis and included within the response plan.
- Stop the movement of any seed that may be infected with the virus, as many viruses are known to be seed-borne (see Table 2).

7.3.2 General procedures for control

Control of sap-sucking insect transmitted viruses is likely to be largely reliant on control of the insect vectors and restrictions on the movement of material that could spread the virus (e.g. live plants, seed (if seed-borne), etc.). Specific control measures will be determined by the CCEPP, however, general procedures include:

- Keep traffic out of affected areas and minimise movement in adjacent areas.
- Adopt best-practice property hygiene procedures to restrict the spread of the pest between fields and adjacent properties.

- After surveys are completed, and permission has been obtained from the Chief Plant Health Manager or the CCEPP, destruction of the infested plant material should follow as plant material can act as a reservoir of the disease.
- On-going surveillance of infected and surrounding areas to ensure the pest is eradicated.
- Do not use seed from infected plants or seed sourced from infected regions for sowing. As many viruses are seed-borne.

7.3.3 Control of infected areas

If an area (such as a portion of a paddock, whole paddock, whole farm, or local area) is found to be infected immediate steps will need to be taken to contain the outbreak. These would likely include the control of the insect vector and the destruction of infected crops, which would otherwise act as a source of infection for surrounding crops. Plant material (such as seed) that could act as a reservoir of the virus should be disposed of by deep burial or incineration on site. The surrounding area should be kept free of host plants including: crops, weeds and pasture species that could act as reservoirs of the virus (or vector).

Particular care must be taken to minimise the transfer of any insect vectors or pant material from the area. All equipment used on the site should be thoroughly cleaned down, with products such as a farm degreaser or a 1% bleach solution and washed down with a pressure cleaner on the affected farm. The clean down procedure should be carried out on a hard surface at a designated wash-down area to avoid recontamination of machinery.

Host plants should not be planted in the infected area for several years, the exact time will depend on the ability of the virus to survive in insect vectors or alternative hosts. Ongoing surveys to ensure that the eradication regime was successful will be determined by the expected survival time of the pathogen in the local environment.

MANAGING VIRUSES

The key points in managing the spread of sap-sucking insect transmitted virus are to:

- Manage the virus by managing either the plant hosts or the insect vector(s).
- Prevent the movement of infected host plants, seedlings and insect-infested plants.
- Control aphids, and other sap-sucking insect vectors, on-farm and in surrounding vegetation using appropriate pesticides, farm management and hygiene practices.
- Minimise handling during the growing season to reduce the mechanical spread of the virus.
- If the virus is seed-borne (such as PStV (Zettler et al., 1993)) ensure any grain that is harvested is not used for seed. Ideally seed should be milled (or otherwise treated) so that it incapable of germination.

MANAGEMENT OF INFECTED CROPS

The movement of insect vectors and live plant material from virus infected crops is a source of infection for healthy crops. Destruction of virus infected crops will control the virus as once dead vectors are unlikely to feed on the crop and spread the virus. However destruction of crops that are heavily infested with insect vectors may cause a mass migration of insects into adjacent crops, unless the vectors are first controlled using a suitable insecticide.

Therefore a possible strategy for the control of a sap-sucking insect transmitted virus would be to first spray a suitable insecticide onto the crop to control the insect vector followed by applications of a suitable herbicide to prevent the crop from acting as a source of future virus infections.

WEED MANAGEMENT

Weeds can serve as alternate hosts of plant viruses and their insect vectors. If weed species are found to be potential hosts of either the virus or vector they will need to be controlled, using a suitable herbicide. Special attention should be paid to weeds along fence lines and road sides adjacent to infected areas or crops as such weeds can allow the virus and/or vector to persist in the area.

7.3.4 Chemical control

Chemical control of viruses is not an option, but suitable chemicals that can be used to effectively control the insect vectors and the virus's host plants would help contain/control the virus.

In the event of an incursion of a sap-sucking insect transmitted virus the vector should be controlled to restrict the spread of the virus. Several chemicals are currently registered in Australia for the control of endemic sap-sucking insects on broadacre crops. A summary of these are described in Table 12. Consult the chemical label before using these products paying attention to all safety information, dosage rates and withholding periods.

Chemicals can also be used to destroy alternative and infected plant hosts in order to slow the spread of the virus. In the case of non-persistent viruses control of the susceptible host plants in the immediate area may help to eradicate the virus. To improve the chances of eradication both host plants and the sap-sucking insect vector(s) should be controlled.

Due to the range of potential host plants specific chemicals will not be detailed in this document, however broad-spectrum herbicides such as Glyphosate and other chemicals may be useful for this purpose. Any chemical used must be approved for that use by the Australian Pesticides and Veterinary Medicines Authority (APVMA) before it can be used in an eradication campaign.

Table 12. *Registered chemicals for the control of sap-sucking insects on Broad acre crops (current as of December 2013)*

Chemical	Pest(s)	Crop(s)
Acetamiprid	Cotton aphid	Cotton
Alpha Cypermethrin	Aphids (*Rhopalosiphum* spp.)	Winter cereals
Amitraz	Cotton aphid	Cotton
Beta-cyfluthrin	Cereal aphids	Cereals
Beta-cyfluthrin	Jassids	Cotton
Chlorantraniliprole with Thiamethoxam	Cotton aphid, Green mirid bug, Yellow mirid, Green peach aphid, Vegetable leafhopper	Cotton
Chlorpyrifos	Spotted alfalfa aphid, Blue-green aphid, Pea aphid	Forage crops - containing lucerne or medics

Chemical	Pest(s)	Crop(s)
Chlorpyrifos	Corn aphid	Sorghum
Chlorpyrifos	Cotton aphid	Cotton
Clothianidin	Cotton aphid, Green Mirid bug, Jassids	Cotton
Cyantraniliprole	Cotton aphid	Cotton
Diafenthiuron	Cotton aphid	Cotton
Diazinon	Blossom thrips	Beans
Diazinon	Lucerne jassid/leafhopper	Lucerne
Diazinon	Spotted alfalfa aphid	Lucerne
Diazinon	Brown plant hopper	Rice
Dimethoate	Jassids	Soybean
Dimethoate	Thrips	Vetch
Dimethoate	Thrips	Sunflower
Dimethoate	Thrips	Pigeon pea
Dimethoate	Jassids	Oilseed crops
Dimethoate	Aphid, Mirid, Thrips	Navy bean
Dimethoate	Aphid, Mirid, Thrips	Mung bean
Dimethoate	Maize leafhopper, Thrips	Maize
Dimethoate	Jassids, Leafhopper and Aphids (including: Blue-green aphid, Spotted alfalfa aphid and Pea aphid)	Lucerne
Dimethoate	Jassids, Aphids, Thrips	Grain legumes
Dimethoate	Jassids, Leafhoppers, Aphids, Mirids	Cotton
Dimethoate	Cereal aphids, Leafhoppers	Cereals
Dimethoate	Leafhopper, thrips	Adzuki bean
Esfenvalerate	*Rhopalosiphum* aphids	Winter cereals
Esfenvalerate	Plague thrips	Lupin
Esfenvalerate	Blue-green aphid	Lucerne
Esfenvalerate	Jassids and Aphids including: Cowpea aphid, Blue-green aphid,	Lentil
Flubendiamide and Thiacloprid	Cotton aphid	Cotton

Chemical	Pest(s)	Crop(s)
Gamma-cyhalothrin	*Rhopalosiphum* aphids	Barley
Gamma-cyhalothrin	Thrips	Canola
Gamma-cyhalothrin	Pea aphid	Lucerne
Gamma-cyhalothrin	*Rhopalosiphum* aphids	Wheat
Imidacloprid	Aphids	Cotton
Imidacloprid	Aphids	Medic pasture
Imidacloprid	Aphids	Clover pasture
Imidacloprid	Aphids	Lupin
Imidacloprid	Aphids	Canola
Imidacloprid	Wheat aphid, Corn aphid	Cereals
Lambda-cyhalothrin	Aphids	Wheat and barley
Lambda-cyhalothrin	Brown mirid, Cotton leafhopper, Vegetable leafhopper	Cotton
Lambda-cyhalothrin	Pea aphid (*Acyrthosiphon pisum*)	Lucerne
Lambda-cyhalothrin	Thrips	Canola
Lambda-cyhalothrin with Thiamethoxam	Aphids including: Green peach aphid	Canola
Lambda-cyhalothrin with Thiamethoxam	Corn aphid, Wheat aphid	Cereals
Maldison	Pea aphid, Spotted alfalfa aphid	Lucerne
Maldison	Jassids, Leafhoppers	Bean (vegetable crops not grain crops)
Maldison	Spotted alfalfa aphid	Pasture
Methidathion	Aphids including: Blue-green aphid, Spotted alfalfa aphid, Lucerne aphid	Lucerne
Methidathion	Cowpea aphid	Lupin
Methomyl	Bean thrips	Legume seed thrips
Methomyl	Pea thrips	Field pea
Methomyl	Common brown leafhopper	Tobacco
Omethoate	Thrips, Mirids, Aphids, Jassids	Cotton
Omethoate	Aphids including: Cowpea aphid	Faba bean
Omethoate	Aphids including: Cowpea aphid	Vetch

Chemical	Pest(s)	Crop(s)
Omethoate	Blue green aphid, Cowpea aphid, Green peach aphid	Lupins
Phorate	Aphids, Jassids, Mirid bugs, Leafhoppers	Cotton
Pirimicarb	Green peach aphid	Canola
Pirimicarb	Cotton aphid and Green peach aphid	Cotton
Pirimicarb	Aphids, including: Pea aphid	Lucerne
Pirimicarb	Green peach aphid, Cowpea aphid	Lupin
Pirimicarb	Aphids (*R. maidis* and *R. padi*)	Cereals
Profenofos	Cotton aphid	Cotton
Pymetrozine	Cotton aphid	Cotton
Spirotetramat	Green peach aphid, Tomato thrips, Western flower thrips	Common bean (vegetable crops not grain crops)
Spirotetramat	Cotton aphid	Cotton
Spirotetramat	Green peach aphid	Pea (vegetable crops not grain crops)
Spirotetramat	Corn aphid	Sweet corn
Sulfoxaflor	Aphids, including Green peach aphid	Barley, wheat
Sulfoxaflor	Aphids including: Green peach aphid	Canola
Sulfoxaflor	Aphids (including: Cotton aphid, Cowpea aphid, Green peach aphid) and Green mirid bug	Cotton
Thiamethoxam	Cotton aphid and Thrips	Cotton

7.3.5 Cultural Control

Cultural controls that may assist in minimising the populations of sap-sucking insects and any viruses they vector include:

- Varying the planting time

  Planting time is known to have an effect on the aphid pressure on crops such as faba beans. For example, New South Wales Department of Primary Industries (2012) suggests that planting faba beans too early in the season increases the risk of aphid damage.

- Planting trap crops

  Trap or barrier crops are another cultural method of controlling insect transmitted viruses. These work by planting a second crop within or near the main crop. The second crop (a non-host crop of the virus) is there to attract the insect vectors away from the main crop where they can then be controlled (Heikki and Hokkanen 1991; Shelton and Badenes-Perez 2006).

- Plant spacing

  The row spacing of the host crop can also have an effect on the aphid population. A'Brook (1968) found that Cowpea aphid (*Aphis craccivora*) and Cotton aphid (*Aphis gossypii*) were more often caught in traps (positioned ~900mm above ground level) when a peanut crop was widely spaced than when plants were more closely spaced, which he suggests is a possible reason for the lower incidence of Rosette disease (an exotic aphid transmitted virus, see Table 2) in closely spaced peanut crops.

7.3.6 Host-Plant Resistance

Host plant resistance/tolerance to viruses would be an effective management tool for farmers and agronomists, as plant resistance offer a low cost way of managing the impact of plant pests and pathogens.

There are currently no lines of chickpea resistant to CpCDV or CpCSV. However there is a degree of tolerance in peanuts to PStV (Higgins et al., 2004) and many other viruses are currently controlled by planting resistant plant varieties.

7.3.7 Biological control of vectors

Plant viruses cannot be directly controlled by antagonistic organisms. Instead the insect vectors or the plant hosts need to be controlled to manage or eradicate the virus.

Aphids, leafhoppers, thrips and other sap-sucking insects can be controlled biologically using various antagonistic organisms. These include the use of wasps, lacewings, flies, predatory bugs and predatory beetles. Some fungi can also be used to control the insects that transmit plant viruses.

Some of the biological controls that have been used to control aphids and leafhoppers in Australia and overseas are described in Table 13.

Table 13 *Biological controls used on aphids and leafhoppers in Australia and overseas*

Insect pest controlled	Biological control	Life form of biological control agent	Reference
Acyrthosiphon pisum	*Aphelinus abdominalis*	Wasp	Waterhouse and Sands (2001)
Acyrthosiphon pisum	*Aphelinus asychis*	Wasp	Waterhouse and Sands (2001)
Acyrthosiphon pisum	*Aphidius eadyi*	Wasp	Waterhouse and Sands (2001)
Acyrthosiphon pisum	*Aphidius ervi*	Wasp	Waterhouse and Sands (2001)
Acyrthosiphon pisum	*Aphidius pisivorus*	Wasp	Waterhouse and Sands (2001)
Acyrthosiphon pisum	*Aphidius smithi*	Wasp	Waterhouse and Sands (2001)
Acyrthosiphon pisum	*Aphidius urticae group*	Wasp	Waterhouse and Sands (2001)
Acyrthosiphon pisum	*Pandora neoaphidis*	Fungus	Waterhouse and Sands (2001)

Insect pest controlled	Biological control	Life form of biological control agent	Reference
Aphis craccivora	*Aphelinus gossypii*	Wasp	Waterhouse and Sands (2001)
Aphis craccivora	*Aphelinus mariscusae*	Wasp	Waterhouse and Sands (2001)
Aphis craccivora	*Aphidius colmani*	Wasp	Waterhouse and Sands (2001)
Aphis craccivora	*Aphidius similis*	Wasp	Waterhouse and Sands (2001)
Aphis craccivora	*Diaeretiella rapae*	Wasp	Waterhouse and Sands (2001)
Aphis craccivora	*Lysiphlebus fabarum*	Wasp	Waterhouse and Sands (2001)
Aphis craccivora	*Lysiphlebus testaceipes*	Wasp	Waterhouse and Sands (2001)
Aphis craccivora	*Trioxys indicus*	Wasp	Waterhouse and Sands (2001)
Aphis glycines	*Harmonia axyridis*	Beetle	Garginer and Landis (2007); Fox et al., (2004)
Aphis glycines	*Aphidoletes aphidimyza*	Fly	Garginer and Landis (2007)
Aphis glycines	*Chrysoperla carnea*	Lacewing	Garginer and Landis (2007)
Aphis glycines	*Orius insidiosus*	Bug	Fox et al., (2004)
Aphis glycines	*Leucopis* spp.	Fly	Fox et al., (2004)
Aphis gossypii	*Aphelinus gossypii*	Wasp	Waterhouse and Sands (2001)
Aphis gossypii	*Aphelinus humilis*	Wasp	Waterhouse and Sands (2001)
Aphis gossypii	*Aphidius colemani*	Wasp	Waterhouse and Sands (2001)
Aphis gossypii	*Lysiphlebus fabarum*	Wasp	Waterhouse and Sands (2001)
Aphis gossypii	*Lysiphlebus testaceipes*	Wasp	Waterhouse and Sands (2001)
Aphis gossypii	*Neozygites fresenii*	Fungus	Waterhouse and Sands (2001)
Hysteroneura setariae	*Diaeretiella rapae*	Wasp	Miller et al., (2002)
Hysteroneura setariae	*Lysiphlebus testaceipes*	Wasp	Miller et al., (2002)
Myzus persicae	*Aphidius colmani*	Wasp	Waterhouse and Sands (2001)
Myzus persicae	*Aphidius similis*	Wasp	Waterhouse and Sands (2001)
Myzus persicae	*Ephedrus persicae*	Wasp	Waterhouse and Sands (2001)
Neolimnus aegyptiacus	None recorded	N/A	N/A
Orosius albicinctus	*Brumus suturalis*[5]	Beetle	Bindra and Singh (1970)

[5] Fed on 1st and 2nd instar larvae in laboratory. No natural enemies found in the field (Bindra and Singh 1970).

Insect pest controlled	Biological control	Life form of biological control agent	Reference
Orosius albicinctus	*Aspergillus flavus*[6]	Fungus	Bindra and Singh (1970)
Orosius albicinctus	*Cladosporium tenusemium*[6]	Fungus	Bindra and Singh (1970)
Orosius orientalis	None recorded. Primarily controlled with insecticides	N/A	Trebicki (2010)

[6] Fungi only weakly parasitic to this pest (Bindra and Singh 1970).

8 Course of action

Additional information is provided by the IPPC (1998b) in Guidelines for Pest Eradication Programmes. This standard describes the components of a pest eradication programme which can lead to the establishment or re-establishment of pest absence in an area. A pest eradication programme may be developed as an emergency response measure to prevent establishment and/or spread of a pest following its recent entry (re-establish a Pest Free Area) or a measure to eliminate an established pest (establish a Pest Free Area). The eradication process involves three main activities: surveillance, containment, and treatment and/or control measures.

8.1 Destruction strategy

8.1.1 Destruction protocols

General protocols:

- No plant material should be removed from the infested area unless part of the disposal or sampling procedure.
- Disposable equipment, infested plant material or soil should be disposed of by autoclaving, high temperature incineration or deep burial (preferably on site).
- Any equipment or plant material removed from the site for disposal should be securely contained.
- All vehicles and farm machinery that enter the infected field should be thoroughly washed, preferably using a detergent, farm degreaser or a 1% (available chlorine) bleach solution.

8.1.2 Decontamination protocols

If decontamination procedures are required, machinery, equipment and vehicles in contact with infected plant material or soil or working within the Quarantine Area, should be washed to remove soil and plant material using high pressure water or scrubbing with products such as a farm degreaser or a 1% bleach solution in a designated wash down area. Disinfection and decontamination guidelines are available as a supporting document of PLANTPLAN (Plant Health Australia, 2013) (**www.planthealthaustralia.com.au/wp-content/uploads/2013/12/Guidelines-Disinfection-and-decontamination.pdf**).

General guidelines for wash down areas are as follows:

- Located away from crops or sensitive vegetation.
- Readily accessible with clear signage.
- Access to fresh water and power.
- Mud free, including entry and exit points (e.g. gravel, concrete or rubber matting).
- Gently sloped to drain effluent away.
- Effluent must not enter water courses or water bodies.

- Allow adequate space to move larger vehicles.
- Away from hazards such as power lines.
- Waste water, soil or plant residues should be contained
- Disposable overalls and rubber boots should be worn when handling infected soil or plant material in the field. Footwear and clothes in contact with infected soil or plant material should be disinfected at the site or double-bagged to remove for cleaning.
- Skin and hair in contact with infested plant material or soil should be washed.

In the event of an incursion of a sap-sucking insect transmitted virus, additional or modified procedures may be required for the destruction of the pest. Any sterilisation procedure must be approved for use in the endorsed Response Plan.

8.1.3 Plants, by-products and waste disposal

- Any soil or infected plant material removed from the infected site should be destroyed by (enclosed) high temperature incineration, autoclaving or deep burial.
- As insects capable of transmitting viruses are small and can be accidentally spread, plant debris from the destruction zone must be carefully handled and transported.
- Infested paddocks should remain free of susceptible host plants (including weeds and volunteer plants) (see Section 5.2.3, 5.3.3 and 5.4.3 for hosts of PStV, CpCDV and CpCSV) until the area has been shown to be free from the virus.
- If the virus is seed-borne seed from the infected paddock will need to be collected and incinerated or double bagged and deep buried in an approved site (preferably away from host plants).

8.2 Containment strategies

For some exotic pest incursions where eradication is considered impractical, containment of the virus may be attempted to prevent or minimise its spread and impact on other areas. The decision to eradicate or contain the virus will be made by the National Management Group based on scientific and economic advice.

8.3 Quarantine and movement controls

Consult PLANTPLAN (Plant Health Australia 2013) for administrative details and procedures.

8.3.1 Quarantine priorities

- Plant material (particularly seed if the virus is known to be seed-borne) from the infected site is to be subject to movement restrictions.
- Machinery, equipment, vehicles and disposable equipment in contact with infested plant material, or present in close proximity to the site of infestation to be subject to movement restrictions.

8.3.2 Movement controls

If Restricted or Quarantine Areas are practical, movement of equipment or machinery should be restricted and movement into the area only allowed by permit.

Movement of people, vehicles and machinery, to and from affected farms, must be controlled to ensure that infected soil or plant debris (including seed) is not moved between properties. This can be achieved through the following; however specific measures must be endorsed in the Response Plan:

- Signage to indicate quarantine area and restricted movement into and within these zones.
- Fenced, barricaded or locked entry to quarantine areas.
- Movement of equipment, machinery, plant material or soil by permit only. Therefore, all non-essential operations in the area or on the property should cease.
- Where no dwellings are located within these areas, strong movement controls should be enforced.
- Where dwellings and places of business are included within the Restricted and Control Areas movement restrictions are more difficult to enforce, however limitation of contact with infested plants should be enforced.
- Clothing and footwear worn at the infected site should either be decontaminated or should not leave the farm until thoroughly disinfected, washed and cleaned.
- Residents should be advised on measures to minimise the inadvertent transport of insects (that could vector the virus) from the infested area to unaffected areas.
- Plant material or plant products must not be removed from the site unless part of an approved disposal or sampling procedure.
- All machinery and equipment should be thoroughly cleaned down with a high pressure cleaner (see Section 8.1.2) or scrubbing with products such as a farm degreaser or a 1% bleach (available chlorine) solution, prior to leaving the affected area.
- Seed from the affected site should not be used for planting new crops, especially if the virus is known to be seed-borne.
- Hay, stubble or trash should not be removed from the site, as these materials could inadvertently spread insect vectors (carrying the virus) between areas.

8.4 Zoning

The size of each quarantine area will be determined by a number of factors, including the location of the incursion, biology of the pest, climatic conditions and the proximity of the infested property to other infested properties. This will be determined by the Response Agency during initial containment efforts and during the development of the Response Plan. Further information on quarantine zones in an Emergency Plant Pest (EPP) incursion can be found in Section 4.1.4 of PLANTPLAN (Plant Health Australia 2013). These zones are outlined below and in Figure 13.

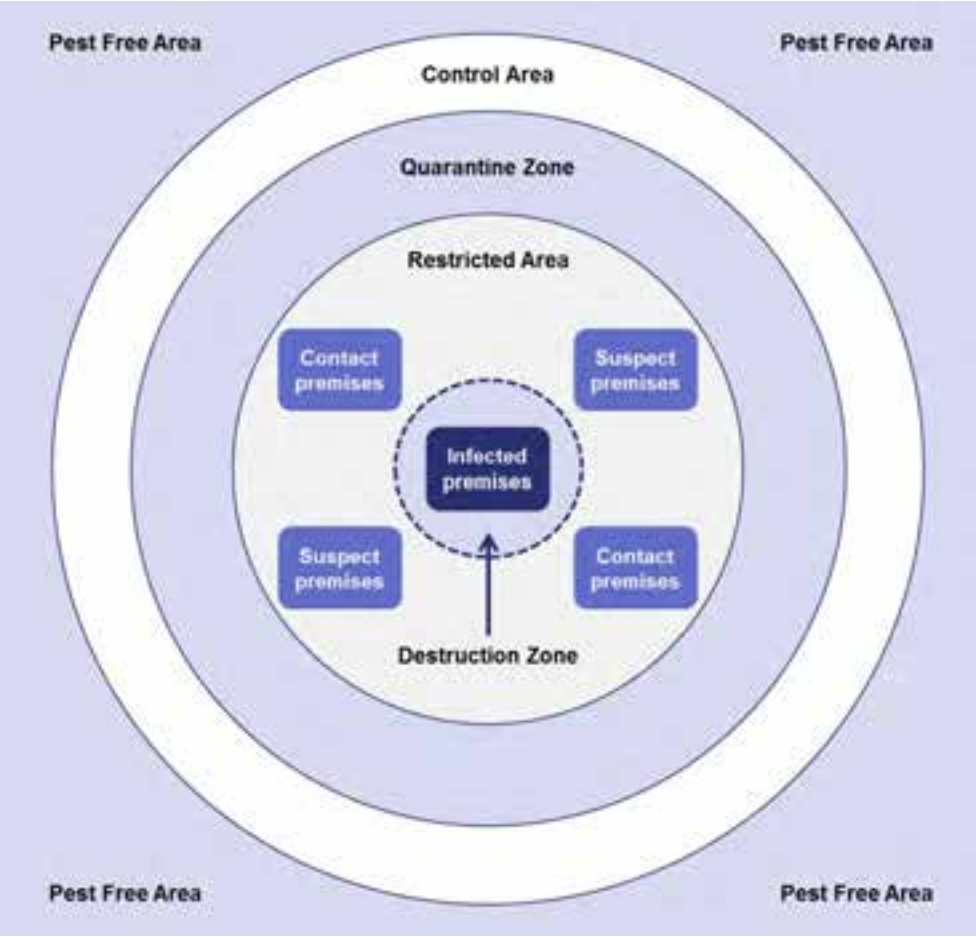

Figure 13. *Schematic diagram of quarantine zones used during an EPP incursion (not drawn to scale)*

8.4.1 Destruction Zone

The size of the destruction zone (i.e. zone in which the pest and all host material is destroyed) will depend on the ability of the pest to spread, distribution of the pest (as determined by delimiting surveys), time of season (and part of the vectors life cycle being targeted) and factors which may contribute to the natural or assisted spread of the pest.

If destruction of hosts is considered, the entire crop should be destroyed after the extent of infection has been established. The delimiting survey will determine whether or not neighbouring host crops are infected and need to be destroyed.

The Destruction Zone will usually be the entire crop but may be the entire farm or contiguous areas of management if spread is likely to have occurred prior to detection.

If the movement of the pest to adjacent crops appears likely, they will also need to be destroyed.

Particular care needs to be taken to ensure that plant material or soil is not moved into surrounding areas. Where possible, destruction should take place in dry conditions to limit the movement of mud and plant material between areas.

8.4.2 Quarantine Zone

The Quarantine Zone is defined as the area where voluntary or compulsory restraints are in place for the affected property or properties. These restraints may include restrictions or movement control for the removal of plants, people, soil or contaminated equipment from an infected property.

8.4.3 Buffer Zone

A Buffer Zone may be required depending on the scale and nature of the incident. It is defined as the area in which the pest does not occur but where movement controls or restrictions for removal of plants, people, soil or equipment from this area are still deemed necessary. The Buffer Zone may enclose an infested area (and is therefore part of the Control Area) or may be adjacent to an infested area.

8.4.4 Restricted Area

The Restricted Area is defined as the zone immediately around the infected premises and suspected infected premises. The Restricted Area is established following initial surveys that confirm the presence of the pest. The Restricted Area will be subject to intense surveillance and movement control with movement out of the Restricted Area to be prohibited and movement into the Restricted Area to occur by permit only. Multiple Restricted Areas may be required within a Control Area.

8.4.5 Control Area

The Control Area is defined as all areas affected within the incursion. The Control Area comprises the Restricted Area, all infected premises and all suspected infected premises and will be defined as the minimum area necessary to prevent spread of the pest from the Quarantine Zone. The Control Area will also be used to regulate movement of all susceptible plant species to allow trace back, trace forward and epidemiological studies to be completed.

8.5 Decontamination and hygiene

Decontaminant practices are aimed at eliminating the pathogen thus preventing its spread to other areas.

8.5.1 Decontamination procedures

General guidelines for decontamination and clean up:

- Refer to Disinfection and decontamination guidelines available as a supporting document of PLANTPLAN (Plant Health Australia, 2013) (**www.planthealthaustralia.com.au/wp-content/uploads/2013/12/Guidelines-Disinfection-and-decontamination.pdf**).
- Keep traffic out of affected area and minimise it in adjacent areas.
- Adopt best-practice property hygiene procedures to retard the spread of the virus and vector(s) between fields and adjacent properties.
- Machinery, equipment and vehicles in contact with infested or infected plant material or soil present within the Quarantine Zone, should be washed to remove soil and plant material using high pressure water or scrubbing with products such as a degreaser or a bleach solution in a designated wash down area as described in Section 8.1.2.
- Only recommended materials are to be used when conducting decontamination procedures, and should be applied according to the product label.
- Infested plant material should be disposed of by autoclaving, high temperature (enclosed) incineration or deep burial (on site).

8.5.2 General safety precautions

For any chemicals used in the decontamination, follow all safety procedures listed within each MSDS.

8.6 Surveillance and tracing

8.6.1 Surveillance

Detection and delimiting surveys are required to delimit the extent of the outbreak, ensuring areas free of the pest retain market access and appropriate quarantine zones are established.

Initial surveillance priorities include the following:

- Surveying all host growing properties and businesses in the pest quarantine area.
- Surveying all properties and businesses identified in trace-forward or trace-back analysis as being at risk.
- Surveying all host growing properties and businesses that are reliant on trade with interstate or international markets which may be sensitive to the presence of the virus.
- Surveying other host growing properties and backyards.

8.6.2 Survey regions

Establish survey regions around the surveillance priorities identified above. These regions will be generated based on the zoning requirements (see Section 8.4), and prioritised based on their potential likelihood of being infected. Surveillance activities within these regions will either allow for the area to be declared pest free or will help determine the extent of the incursion to allow for further

containment measures. Detailed information regarding surveys for vector(s) and virus infected plant material have been outlined elsewhere in this plan (refer to Section 7.2).

Steps outlined in Table 14 form a basis for a survey plan. Although categorised in stages, some stages may be undertaken concurrently based on available skill sets, resources and priorities.

Table 14. *Phases to be covered in a survey plan*

Phase 1	• Identify properties that fall within the buffer zone around the infected premise. • Complete preliminary surveillance to determine ownership, property details, production dynamics and tracings information (this may be an ongoing action).
Phase 2	• Preliminary survey of host crops on properties in buffer zone establishing points of pest detection.
Phase 3	• Surveillance of an intensive nature, to support control and containment activities around points of pest detection.
Phase 4	• Surveillance of contact premises. A contact premise is a property containing susceptible host plants, which are known to have been in direct or indirect contact with an infected premises or infected plants. Contact premises may be determined through tracking movement of materials from the property that may provide a viable pathway for spread of the pest. Pathways to be considered are: o Items of equipment and machinery which have been shared between properties including bins, containers, irrigation lines, vehicles and equipment. o The producer and retailer of infected material if this is suspected to be the source of the outbreak. o Labour and other personnel that have moved from infected, contact and suspect premises to unaffected properties (other growers, tradesmen, visitors, salesmen, crop scouts, harvesters and possibly beekeepers). o Movement of plant material and soil from controlled and restricted areas. o Storm and rain events and the direction of prevailing winds that result in air-borne dispersal of the vector(s) during these weather events.
Phase 5	• Surveillance of farms, gardens and public land where plants known to be hosts of virus/vector are being grown.
Phase 6	• Agreed area freedom maintenance, post-control and containment.

8.6.3 Post-eradication surveillance

The period of pest freedom sufficient to indicate that eradication of the pest has been achieved will be determined by a number of factors, including growth conditions, the previous level of infection, the control measures applied and the pest biology.

Specific methods to confirm eradication of sap-sucking insect transmitted viruses may include:

- Establishment and monitoring of sentinel plants at the site of infection.
- Sentinel plants are to be grown in containers or small plots at the affected site. Plants are to be grown *in situ* under quarantine conditions and monitored for symptoms of infection.
- If symptoms or virus are detected, samples are to be collected and stored and plants destroyed.

- Surveys comprising of host plant sampling for the virus should be undertaken for a minimum of three years after eradication has been achieved (or as endorsed by the CCEPP).

9 Technical debrief and analysis for stand down

Refer to PLANTPLAN (Plant Health Australia 2013) for further details

The emergency response is considered to be ended when either:

- Eradication has been deemed successful by the lead agency, with agreement by the Consultative Committee on Emergency Plant Pests and the Domestic Quarantine and Market Access Working Group.
- Eradication has been deemed impractical and procedures for long-term management of the disease risk have been implemented.

A final report should be completed by the lead agency and the handling of the incident reviewed.

Eradication will be deemed impractical if, at any stage, the results of the delimiting surveys lead to a decision to move to containment/control.

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11 Appendices

11.1 Appendix 1: Hosts of Peanut stripe virus (*Potyvirus*) as listed on the CAB Compendium (CAB International 2013)

Scientific name	Common name	Family
*Arachis hypogaea**	Peanut	Fabaceae
Calopogonium caeruleum	None	Fabaceae
Centrosema pubescens	Centro	Fabaceae
Crotalaria pallida	Smooth crotalaria	Fabaceae
Desmodium spp.	Tick clovers	Fabaceae
*Glycine max**	Soybean	Fabaceae
Indigofera spp.	Indigo	Fabaceae
*Lupinus albus**	White lupine	Fabaceae
*Medicago sativa**	Lucerne	Fabaceae
Pueraria phaseoloides	Tropical kudzu	Fabaceae
Senna obtusifolia	Sicklepod	Fabaceae
Senna occidentalis	Coffee senna	Fabaceae
Senna tora	Sicklepod	Fabaceae
*Sesamum indicum**	Sesame	Pedaliaceae
Stylosanthes spp.	Pencil flower	Fabaceae
*Vigna radiata**	Mung bean	Fabaceae
*Vigna unguiculata**	Cowpea	Fabaceae

An asterisk (*) indicates those species that are grown as crops in Australia

Limited information is available from CABI on the hosts of CpCDV and CpCSV (see Sections 5.3.3and 5.4.3 for more details on the hosts of these viruses)

11.2 Appendix 2: Standard diagnostic protocols

For a range of specifically designed procedures for the emergency response to a pest incursion refer to Plant Health Australia's PLANTPLAN (**www.planthealthaustralia.com.au/plantplan**).

11.3 Appendix 3: Resources and facilities

Formal diagnostic services for plant pests in Australia are delivered through a network of facilities located in every state and territory. These services are provided by a range of agencies, including state and territory governments, the Australian Government, commercial and private diagnostic laboratories, museums, CSIRO and universities. A current listing of these facilities can be found at **www.npbdn.net.au/resource-hub/directories/laboratory-directory**

The national network is supported by the Subcommittee on Plant Health Diagnostic Standards (SPHDS), which was established to improve the quality and reliability of plant pest diagnostics in Australia. SPHDS also manages the production of National Diagnostic Protocols.

For more information on the diagnostic services, or to identify an appropriate facility to undertake specific pest diagnostic services, refer to **www.npbdn.net.au** or contact the SPHDS Executive Officer on **SPHDS@daff.gov.au**

11.4 Appendix 4: Communications strategy

A general Communications Strategy is provided in Section 4.1.5 of PLANTPLAN (Plant Health Australia, 2013).

11.5 Appendix 5: Market access impacts

Within the Department of Agriculture Manual of Importing Country Requirements (MICoR) database (**www.daff.gov.au/micor/plants/**) export of some material may require an additional declaration regarding freedom from the virus. Should exotic sap-sucking insect transmitted viruses be detected or become established in Australia, some countries may require specific declarations. Latest information can be found within MICoR, using a search for the particular virus.

The Department of Agriculture MICoR database was searched in December 2013 for current trade restrictions relating to the three viruses used as examples in this contingency plan. No countries were identified on the Department of Agriculture MICoR database as having trade restrictions regarding Chickpea chlorotic dwarf virus or Chickpea chlorotic stunt virus. However Botswana has some restrictions relating to the presence of Peanut stripe virus (see Table 15).

Table 15 *Countries identified on the Department of Agriculture MICoR database that have trade restrictions regarding Peanut stripe virus*

Country	Commodity	Requirements/restrictions
Botswana	*Vigna* spp. seed	Declaration that Peanut stripe virus is not known to occur in Australia
Botswana	*Phaseolus* spp. seed	Declaration that Peanut stripe virus is not known to occur in Australia