

**INDUSTRY BIOSECURITY PLAN
FOR THE GRAINS INDUSTRY**

Generic Contingency Plan

Exotic soil-borne pathogens affecting the grains industry

Specific examples detailed in this plan:

Late wilt of maize (*Harpophora maydis*)

and

Downy mildew of sunflower (*Plasmopara halstedii*)

Plant Health Australia

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**Grains Research &
Development Corporation**



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1 Purpose and background of this contingency plan

Developing a generic contingency plan framework for groups of exotic pests will ensure the industry is better prepared for new pest incursions. This generic contingency plan framework is designed to assist the grains industry for an incursion of a soil-borne fungal pathogen that may not already be covered by a pest specific contingency plan. As most soil-borne pathogens share a common behaviour in terms of their ability to spread to new areas by the movement of soil this generic framework has implications for the management of this group of soil-borne pests.

This contingency plan framework provides background information on the pest biology, available control measures and other relevant information to assist with preparedness for an incursion into Australia of soil-borne fungal pathogens that could potentially impact on the grains industry. Two soil-borne pathogens have been used as examples of pests could potentially enter Australia. It should be noted that some soil-borne pathogens are already present in Australia and that endemic pathogens are not considered in this contingency plan framework.

The contingency plan provides guidelines and options for steps to be undertaken and considered when developing a Response Plan for an incursion of an exotic soil-borne pathogen. 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 background, disease cycle, host range, distribution and symptoms of two specific soil-borne pathogens are given as examples, with the emphasis of this document on the management options in the event of an exotic soil-borne pathogen incursion into 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 Australia's most widely planted grain crop and is grown in all areas of the grain belt (Figure 1).

The grains industry consists of 25 leviable crops. Most, if not all, are susceptible to one or more species of soil-borne pathogens.

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. With this reliance on exports, maintaining our current plant health status through appropriate biosecurity measures is essential.

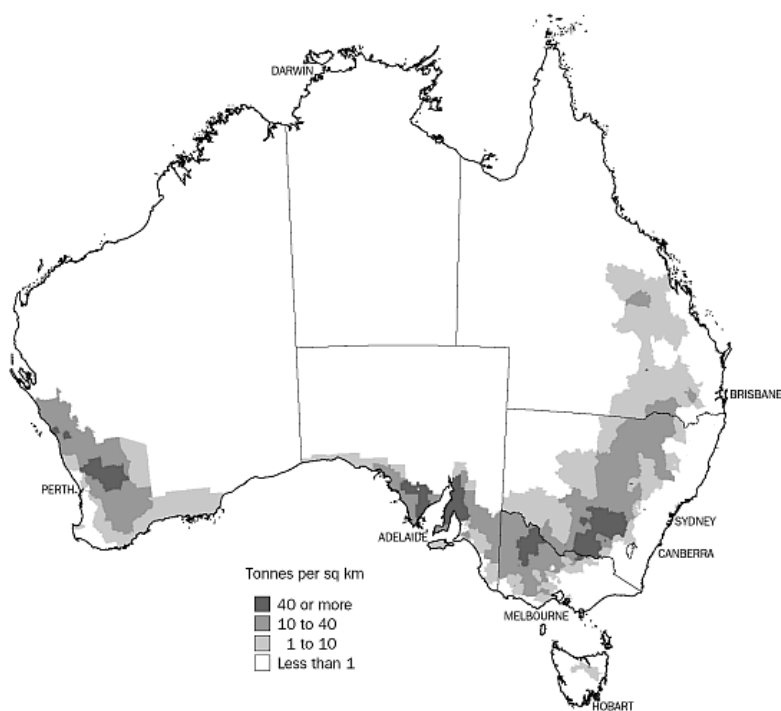


Figure 1 Map of wheat producing regions in Australia (i.e. the grain belt). (Source ABS 2007)

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 soil-borne fungal pathogen. The notification process is described in Figure 2.

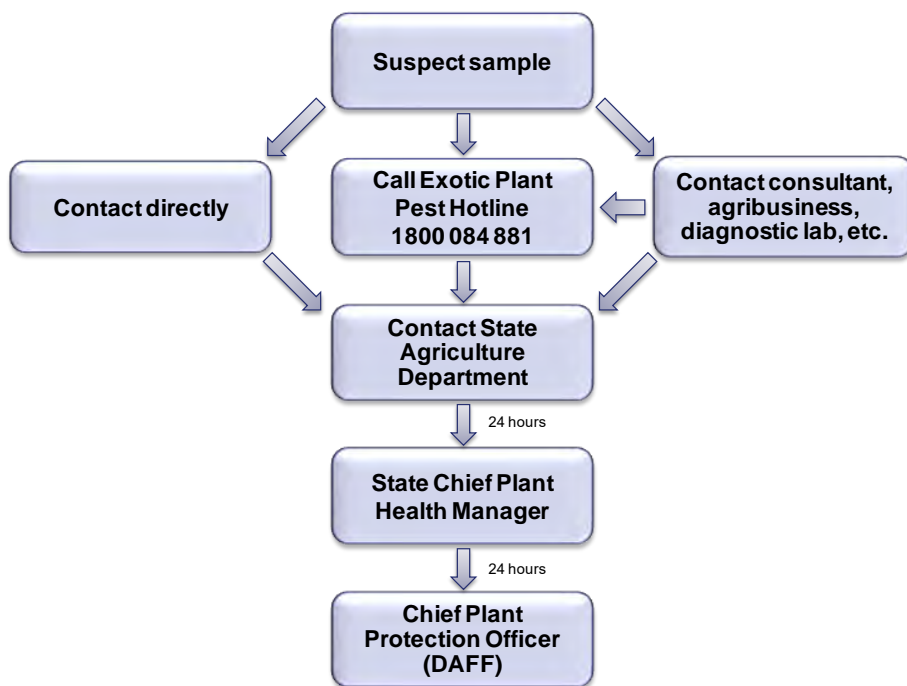


Figure 2. Notification process for the reporting of suspect pests

3 Eradication or containment decision matrix

The decision to eradicate should be based on the potential economic impact of host damage resulting from the introduction of a soil-borne pathogen, the cost of eradication and technical feasibility. Eradication costs must factor in long term surveys to prove the success of the eradication program.

Before pest free status can be declared the exact number of years with no detection of the pathogen will depend on the pathogen’s biology and the survival ability of the inoculum. For example as the inoculum of *P. halstedii* (Downy mildew of sunflower) can survive in the soil for up to ten years (Vear et al., 2008) a minimum of ten years would be required with no detection of the pathogen before a pest free status may be declared. While, the inoculum of *H. maydis* (Late wilt of maize) only remains viable for 12-15 months (United States Department of Agriculture 2011; Sabet et al., 1970) and therefore a much shorter detection free period would occur before an area could be declared as being pest free.

No specific eradication matrix has been determined for soil-borne pathogens, however the general decision process as outlined in Figure 3 and Table 1 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.

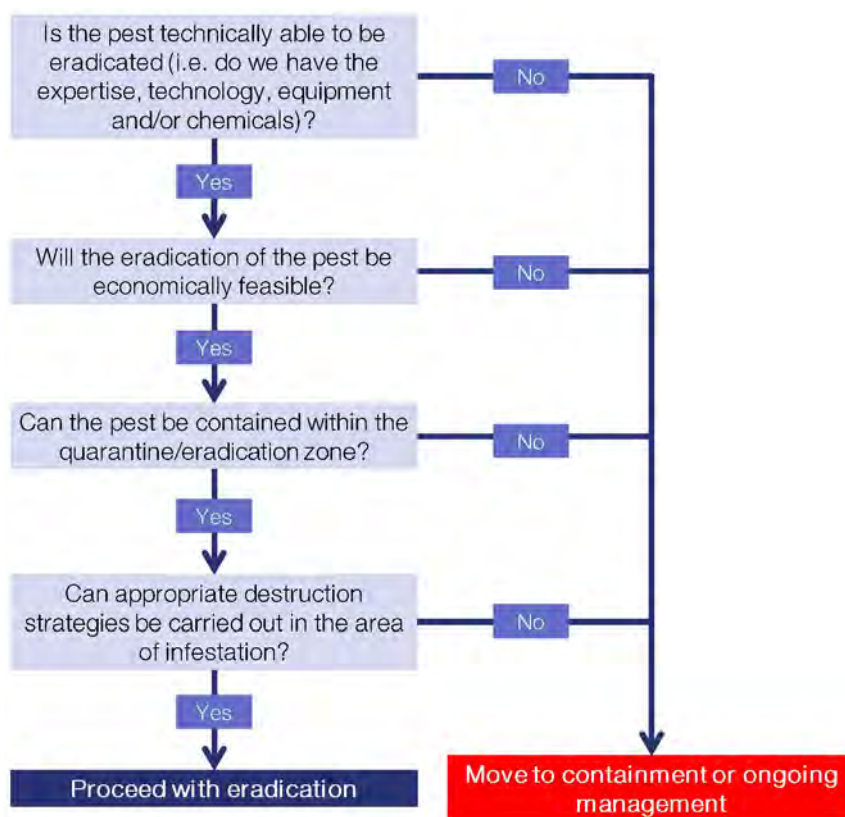


Figure 3. Decision outline for the response to an exotic pest incursion

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

Factors favouring eradication	Factors favouring alternative action
<ul style="list-style-type: none"> • Cost/benefit analysis shows significant economic or amenity loss to industry or the community if the organism establishes. • Physical barriers and/or discontinuity of hosts between production districts. • Cost effective control difficult to achieve (e.g. limited availability of protectant or curative treatments). • The generation time, population dynamics and dispersal of the organism favour more restricted spread and distribution. • Pest biocontrol agents not known or recorded in Australia. • Vectors discontinuous and can be effectively controlled. • Outbreak(s) 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. 	<ul style="list-style-type: none"> • Cost/benefit analysis shows relatively low economic or environmental impact if the organism establishes. • Major areas of continuous production of host plants. • Cost effective control strategies available. • Short generation times, potential for rapid population growth and long distance dispersal lead to rapid establishment and spread. • Widespread populations of known pest biocontrol agents present in Australia. • Vectors unknown, continuous 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.

4 Pest information/status – Exotic soil-borne fungal pathogens that affect grain crops

4.1 Background

There are a number of exotic soil-borne fungal pathogens identified in the Grains Industry Biosecurity Plan (Plant Health Australia 2009, currently under review) that can be spread by the movement of soil between countries, regions and farms (see Table 2). Some genera can also be spread by the movement of seed, spread over short distances by the wind, or rain splashed spores (see Table 3).

The two species used as examples in this contingency plan have been selected to illustrate the management options available in the event of an incursion of an exotic soil-borne pathogen. Even though there are some differences between genera most soil-borne pathogens are spread in a similar manner in the soil or as air-borne spores.

Most soil-borne pathogens will be controlled in a similar manner, however specific chemicals, application rates, biological controls, etc. are likely to vary between species and will have to be considered on a case by case basis. Details such as the general procedures for control (Section 1.1.1), sampling protocols (Section 6.2), quarantine and movement controls (Section 7.3), zoning requirements (Section 7.4) and other components of this contingency plan will be the same for all exotic soil-borne pathogens should they enter Australia and impact on the grains industry.

Table 2 Some exotic soil-borne fungal pathogens identified in the Grains Industry Biosecurity Plan (Plant Health Australia 2009)

Common name	Scientific name	Overall risk ¹
Leaf spot	<i>Alternaria humicola</i> ²	High
Fusarium wilt	<i>Fusarium avenaceum</i> f. sp. <i>fabae</i>	Medium
Fusarium wilt	<i>Fusarium oxysporum</i> f. sp. <i>ciceris</i> ³	High
Fusarium wilt	<i>Fusarium oxysporum</i> f. sp. <i>conglutinans</i> race1 ⁴	Low-medium
Lentil wilt	<i>Fusarium oxysporum</i> f. sp. <i>lentis</i> ³	Medium
Lupin wilt	<i>Fusarium oxysporum</i> f. sp. <i>lupini</i> ³	Low
Late wilt of maize	<i>Harpophora maydis</i> (syn. <i>Acremonium maydis</i> ; <i>Cephalosporium maydis</i>) ⁵	High
Cephalosporium stripe	<i>Hymenula cerealis</i>	Very low
Downy mildew of sorghum	<i>Peronosclerospora sorghi</i> ⁶	High
Chickpea root rot	<i>Phacidiopycnis padwickii</i>	Negligible
Brown stem rot	<i>Phialophora gregata</i>	Unknown
Texas root rot	<i>Phymatotrichopsis omnivora</i>	Unknown
Downy mildew of sunflower	<i>Plasmopara halstedii</i> ⁵	High
Sheath spot	<i>Rhizoctonia oryzae</i>	Low
Brown stripe downy mildew	<i>Sclerophthora rayssiae</i>	Unknown
Peanut scab	<i>Sphaceloma arachidis</i>	Low
Dwarf bunt	<i>Tilletia controversa</i> ⁷	Very low
Verticillium wilt	<i>Verticillium dahlia</i> var. <i>longisporum</i> (syn. <i>V. longisporum</i>) ⁸	Medium

¹ Note: when more than one crop is affected by the same species of fungus the highest overall risk has been included in the table.

² Contingency plan available (Murray and Plant Health Australia 2009a).

³ Contingency plan available (Lindbeck and Plant Health Australia 2009).

⁴ Contingency plan available (Kochman and Plant Health Australia 2007).

⁵ Example used in this contingency plan.

⁶ Contingency plan available (Murray and Plant Health Australia 2009b).

⁷ Contingency plan available (Murray and Wright 2007)

⁸ Contingency plan available (Lindbeck and Plant Health Australia 2011).

Table 3 General information of the dispersal of soil-borne pathogens

Genus	General information on dispersal
<i>Alternaria</i> spp. such as <i>A. humicola</i>	Inoculum overwinters in soil, on seeds. Also spread by rain splash or wind between plants.
<i>Fusarium</i> spp. such as <i>F. oxysporum</i>	Inoculum overwinters in soil, crop residues or seeds.
<i>Harpophora</i> spp. such as <i>H. maydis</i>	Inoculum overwinters in plant debris and seed.
<i>Hymenula</i> spp. such as <i>H. cerealis</i>	Inoculum overwinters in crop debris in (or on) the soil. Conidia are soil-borne.
<i>Phacidiopycnis</i> spp. such as <i>P. padwickii</i>	Inoculum occurs in the soil.
<i>Peronosclerospora</i> spp. such as <i>P. sorghi</i>	Inoculum in soil and crop debris. Also spread by the wind.
<i>Phialophora</i> spp. such as <i>P. gregata</i>	Inoculum survives in crop debris and soil.
<i>Phymatotrichopsis</i> spp. such as <i>P. omnivora</i>	Inoculum survives in the soil and crop debris.
<i>Plasmopara</i> spp. such as <i>P. halstedii</i>	Spores can be dispersed in soil, crop debris and occasionally seed. Can also be dispersed by the wind onto the leaves of other plants.
<i>Rhizoctonia</i> spp. such as <i>R. oryzae</i>	Inoculum overwinters in soil and crop debris.
<i>Sclerophthora</i> spp. such as <i>S. rayssiae</i>	Inoculum overwinters in crop debris and in soil. There are some reports of the pathogen being spread with seed. The pathogen's spores are also spread by wind and rain.
<i>Sphaceloma</i> spp. such as <i>S. arachidis</i>	Inoculum overwinters in crop debris in the soil. Can also spread between plants by rain splash. Not spread by seed (Kearney et al., 2002)
<i>Tilletia</i> spp. such as <i>T. controversa</i>	Inoculum overwinters in soil ⁹ or on seed. Pathogen is also spread by machinery (e.g. harvesters) and the wind (but usually only over short distances).
<i>Verticillium</i> spp. such as <i>V. dahlia</i> var. <i>longisporum</i>	Inoculum occurs in the soil and crop debris. Some species such as <i>V. dahlia</i> var. <i>longisporum</i> are spread with seed (Heppner and Heitefuss 1995).

4.2 Generic information on lifecycles

Soil-borne fungal pathogens share a similar lifecycle in that they overwinter (i.e. survive in the absence of host plants) in the soil or in crop debris in the soil (see Table 3). Once hosts are available the pathogen invades the host plant and produces spores, allowing the process to continue.

The exact details of the pathogens lifecycle (such as the spore stages that are involved, speed of the lifecycle, temperatures and conditions required by the pathogen, etc.) are individual to the species concerned and as fungi often have complex lifecycles involving a number of spores and sexual and asexual reproduction stages it is impossible to provide further generic information here. In the event of a pest incursion the lifecycles and biology of each species would need to be considered on an individual basis.

⁹ *T. controversa* can survive 3-10 years (Smilanick et al., 1986)

4.3 Dispersal

Many soil-borne fungal pathogens spread very slowly in the absence of human assistance (such as people moving machinery contaminated with soil, or planting seeds containing inoculum in new areas). Although there are exceptions which can be dispersed to new areas by the movement of air-borne spores (e.g. *Tilletia* spp. and *Plasmopara* spp.) allowing them to spread to new areas faster than other soil-borne pathogens that naturally spread by rain splash or the natural spread of the fungus through the soil (e.g. *Sphaceloma arachidis*).

The ability of exotic soil-borne fungal pathogens to develop and establish in Australia will be determined by the presence of host plants, the movement of inoculum to new areas and the suitability of the environment to the establishment of the pest. In the absence of these it may be difficult for the pest to establish.

4.4 Symptoms

The symptoms caused by soil-borne pathogens vary greatly between species. For example several species (such as *Fusarium* spp. and *Verticilium* spp.) cause the development of wilting symptoms, whereas other such as, *Plasmopara halstedii*, cause stunting and leaf symptoms to develop (see Figure 6 and Figure 7). There are other soil-borne pathogens such as *Tilletia controversa* which cause damage to the developing seeds. As a consequence, this variability shows that no “classic symptoms” of a soil-borne fungal pathogen can be applied to the whole group.

4.5 Sampling

Samples of plants thought to be affected by soil-borne pathogens should be collected and treated as described in Section 6.2. In most cases samples should include infected plants and their roots. Exact details will be decided on a case by case basis.

4.6 General information on the diagnosis of soil-borne pathogens

Currently there are no endorsed diagnostic protocols for soil-borne fungal pathogens that affect the grains industry. For a current list of the endorsed National Diagnostic Protocols see the Subcommittee on Plant Health Diagnostic Standards (SPHDS) website: www.padil.gov.au/sphds

Generally fungi are identified based on the characteristics of their spores or by the use of molecular techniques. Mycological expertise would be required to identify specimens to a species level.

For diagnostic facilities and advisory services that can be utilised in the event of an incursion see Section 10.2 Appendix 2.

4.7 General comments on control

If left unchecked many of the soil-borne pathogens listed in Table 2 could cause significant damage to host crops. Fungicides can be used to control infestations.

The chemical required and rates that it should be applied will need to be determined on a case by case basis and be tailored to the specific species involved. Any chemicals used for the eradication or control of an exotic pest in Australia must be registered for use through the Australian Pesticides and Veterinary Medicines Authority (APVMA). In the event of an incursion, emergency permits may need to be sought. For information regarding this process visit the APVMA website.

Host plant resistance and the use of crop rotations incorporating non-host crops are common ways of managing soil-borne pathogens overseas. For example *P. halstedii* (Downy mildew of sunflower) is often controlled overseas by using resistant sunflower varieties (e.g. Jovic et al., 2010; Vear et al., 2008; Vear et al., 1997). Crop rotations are also widely used. Rotating between host and non-host crops reduces the risk of the pathogen's inoculum building up to damaging levels.

The uses of biological control of soil-borne pathogens are not widely used. Section 6.3.7 provides more information on biological control of the two example soil-borne pathogens.

5 Specific examples of exotic grain affecting soil-borne pathogens

5.1 Pest Details – Late wilt of maize (*Harpophora maydis*)

Common name:	Late wilt of maize; Slow wilt; Black bundle disease
Scientific name:	<i>Harpophora maydis</i> (Samra, Sabet & Hing.) W. Gams 2000
Synonyms:	<i>Acremonium maydis</i> ; <i>Cephalosporium maydis</i>
Taxonomic position:	Kingdom: Fungi Phylum: Ascomycota Class: Ascomycetes Family: Magnaporthaceae Genus: <i>Harpophora</i>

5.1.1 Background

Late wilt of maize (*Harpophora maydis*) is an example of a soil-borne pathogen that has a high overall risk rating to the grains industry (Plant Health Australia 2009).

Late wilt of maize causes significant yield losses in maize, with losses of 40% reported from Egypt (Zeller et al., 2000). The pathogen usually only affects young (< 50 day old) plants (Sabet et al., 1970) infecting the roots before entering the xylem, causing wilt symptoms to develop at the tasseling stage. This pathogen occurs in southern Europe, North Africa and India and as yet has not been reported in Australia. There are 17 known strains of this pathogen (Saleh and Leslie 2004), which has

implications for its management as different strains of the pathogen may respond to chemicals and host plants differently.

5.1.2 Disease cycle

H. maydis is only known to reproduce asexually (Saleh and Leslie 2004). The disease cycle is shown in Figure 4. The fungus survives in the soil or on crop debris as sclerotia or mycelia. Once in contact with the roots of a susceptible host the sclerotia germinate to produce mycelia. The fungus produces hyphae which at first grow epiphytically on the roots, penetrating the roots then growing inter- and intra-cellularly. The conidia and hyphae of the fungus grow upwards through the stem, blocking vessels resulting in wilting at the tasseling stage (i.e. BBCH 51-59) (Pecsi 1996; Zeller et al., 2002). In some cases seed from infected plants may also act as a source of inoculum (Johal et al., 2004; Michail et al., 1999).

This pathogen has been shown to survive for 12-15 months in soil with spore survival restricted to the top 20 cm of soil (USDA 2011; Sabet et al., 1970).

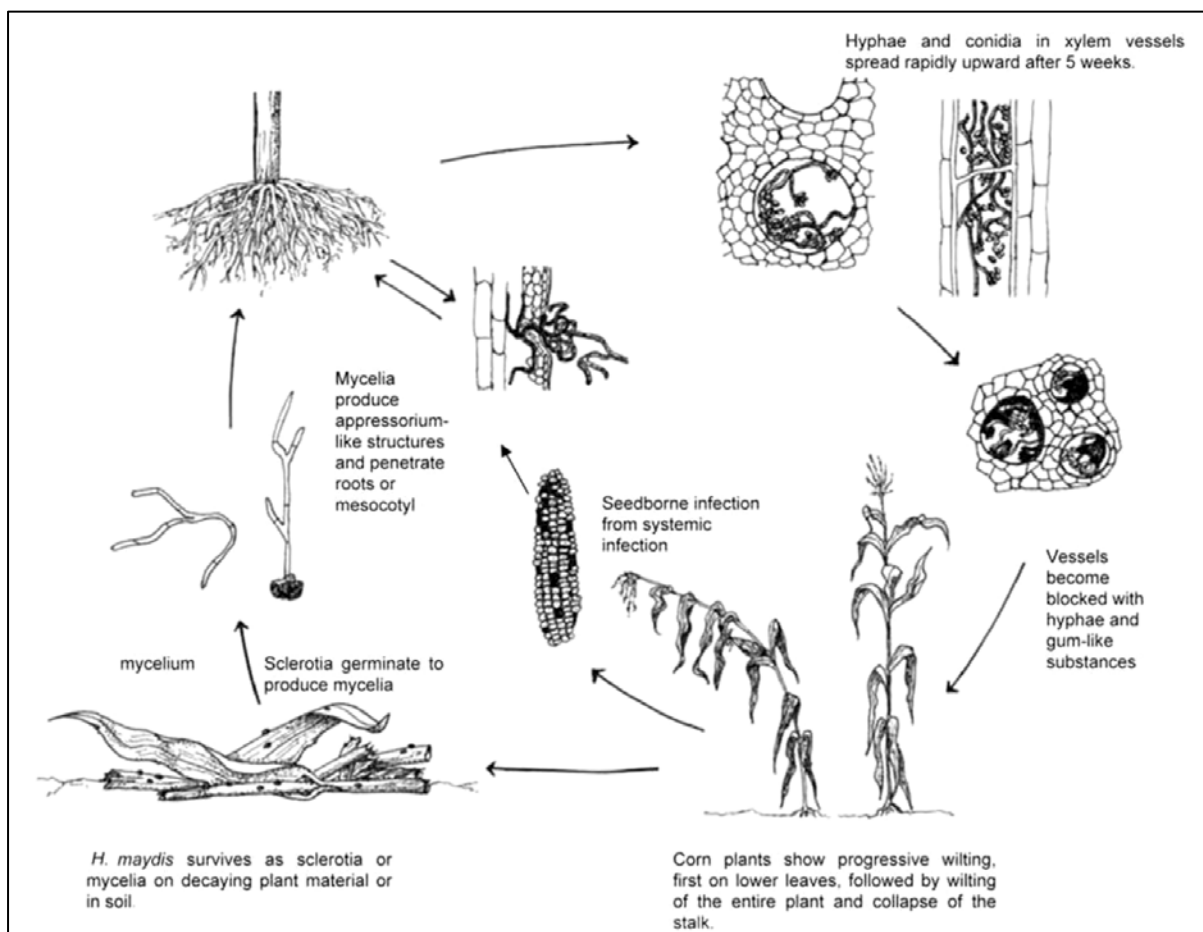


Figure 4 Disease cycle of *H. maydis*. (Source: Johal et al., (2004))

5.1.3 Dispersal

This pathogen is soil-borne (Saleh and Leslie 2004) and can also be present in plant debris and seed (Johal et al., 2004; Saleh et al., 2003). Therefore it can be spread by the movement of soil from infected areas to non-infected areas on machinery, vehicles, footwear, or other goods or by the movement of infected plant material (including seeds).

Therefore in the event of a pest incursion restrictions should be placed on the movement of soil, plant debris and seed (including material adhering to machinery) to limit the spread of the pathogen to new areas.

5.1.4 Host range

Late wilt of maize is principally a pest of maize, *Zea mays* (Payak et al., 1970; Sabet 1970; Molinero-Ruiz et al., 2010). However the pathogen also affects cotton, *Gossypium hirsutum* (Sabet et al., 1966) and in Egypt the pathogen has been found attacking the roots of lupin, *Lupinus termis* (syn. *L. albus*) (Abd-El-Kareem et al., 2004) (*L. albus* is a commonly grown grain lupin in Australia). No other host plants have been recorded for this pathogen in the scientific literature.

5.1.5 Current geographic distribution

This pathogen occurs in maize growing regions of Northern Africa, southern Europe and India, as shown in Table 4. It has not been reported from Australia.

Table 4 Geographic distribution of *H. maydis*

Country	Reference
Egypt	Saleh et al., (2003)
Hungary	Pecsi (1996)
India	Payak et al., (1970)
Israel*	Bergstrom et al., (2008)
Italy*	Bergstrom et al., (2008)
Kenya*	Ward and Bateman (1999)
Portugal	Molinero-Ruiz et al., (2010)
Romania*	Bergstrom et al., (2008)
Spain	Molinero-Ruiz et al., (2010)

* Note: the reports from Italy, Romania, Israel and Kenya have not been confirmed but are suspected. For example Ward and Bateman (1999) describe finding symptoms similar to *H. maydis* in Kenya but did not formally identify the causal agent.

5.1.6 Potential geographic distribution in Australia

As described in Table 4 this pathogen has a wide geographic distribution and is able to survive in a range of climates. The minimum temperature that the fungus has been found to grow at was 6 °C (Pecsi and Nemeth 1998; Magarey et al., 2008), while the maximum temperature that growth occurred was found to be between 35 and 40 °C (Singh and Siradhana 1985; Magarey et al., 2008).

Magarey et al., (2008) created a map of the pathogens potential distribution based on world climate data and the number of favourable days different areas have for the development of the pathogen. The researchers noted that most of the USA, Mexico, northern and central South America, Africa, southern Asia and most of Australia (including northern NSW and Queensland maize growing regions) have suitable climatic conditions for the pathogen during the spring/summer period, which is when the pathogen is most likely to infect host plants.

Given the current geographic distribution and the range of temperatures that this pathogen is able to survive in, as well as the maps produced by Magarey et al., (2008) it is likely that all maize growing regions of Australia could be affected by *H. maydis*, with areas in northern NSW and Queensland being more severely affected (i.e. having more favourable days per year) than the irrigation areas of south-western NSW (see Figure 5).

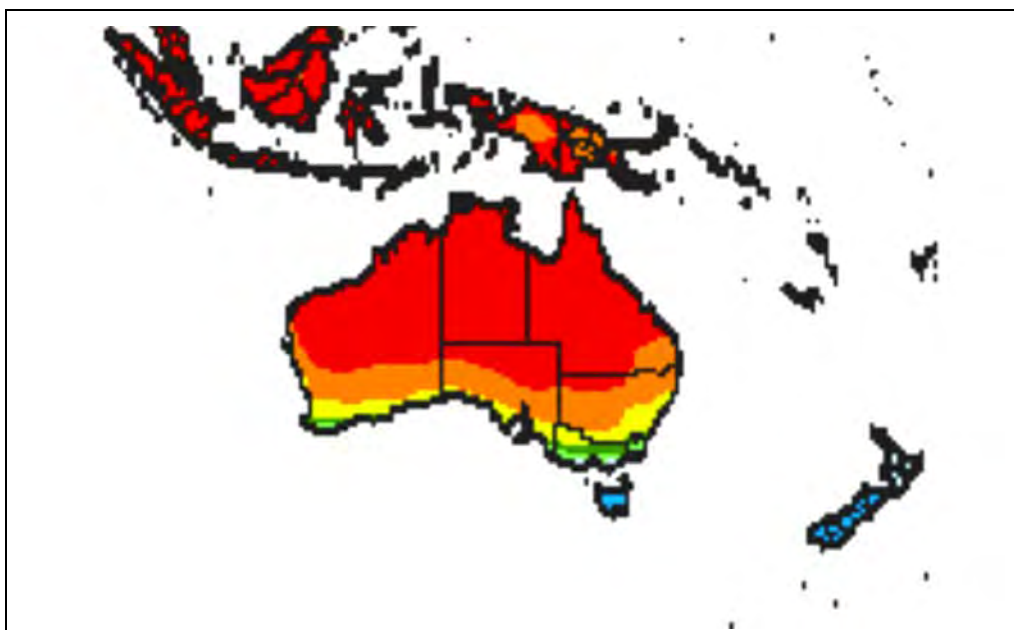


Figure 5 Suitability of the climate for *H. maydis*. Note: Red, orange and yellow areas have more favourable days than Hungary, a country where the disease has been previously recorded, and therefore are potential areas where the pathogen could survive. Modified from: Magarey et al., (2008)¹⁰

¹⁰ Note: Red areas have more favourable days than orange areas, which have more favourable days than the yellow areas of the map. Green and blue areas of the map have fewer favourable days than Hungary and are therefore considered to be the areas of least risk of supporting the pathogen.

5.1.7 Symptoms

Late wilt of maize causes the roots of infected plants to become red stained, however the above-ground parts of the plant only develop obvious symptoms as tasseling occurs (BBCH 51-59). Discoloured streaks occur on leaves and stems and the plant wilts then dies. When cut in half the centre of infected stalks will have a red-brown discolouration (Bergstrom et al., 2008).

The effect of *H. maydis* on cotton was studied by Sabet et al., (1966). They found that the pathogen is associated with the formation of dark red lesions on the roots of the cotton plant but as the plant ages the root hardens and the lesions disappear. The pathogen has also been associated with an increase in the number of lateral roots produced by the cotton plant.

In lupins the pathogen has been associated with significant damping off and stunting (Bergstrom et al., 2008), which suggests that this pathogen could also impact on Australia's lupin industry.

5.1.8 Diagnostic information

Currently there is not an endorsed National Diagnostic Protocol for Late wilt of maize.

For diagnostic facilities and advisory services that can be utilised in the event of an incursion see Section 10.2 Appendix 2.

5.1.8.1 MORPHOLOGICAL AND PHYSIOLOGICAL DIAGNOSIS

Conidia, conidiophores and hyphae morphology together with the observation of wilting symptoms can assist in the preliminary identification of this pathogen. USDA (2011) describes the conidia as sickle shaped and the hyphae as hyaline when young but become darker with age. CABI (2012) describe conidiophores as between 60 and 250 µm in length and conidia as mostly single celled and 3.5 - 14.0 by 3.5 µm in size.

5.1.8.2 PCR

Polymerase Chain Reaction (PCR) is a rapid, specific, and sensitive test that can be used to detect and diagnose Late wilt of maize. Saleh and Leslie (2004) used PCR to test the relationships of *H. maydis* to other species in the *Gaeumannomyces-Harpophora* species complex.

Amplified Fragment Length Polymorphism (AFLP) has also been used to look at the genetic relationships between *H. maydis* from different areas of Egypt (Saleh et al., 2003).

No commercially available tests for *H. maydis* are currently available.

5.1.9 Pest risk analysis – Late wilt of maize

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

5.1.9.1 ENTRY POTENTIAL

Rating: High

H. maydis is a soil-borne pathogen (Saleh and Leslie 2004) and can also be present in plant debris and seed (Johal et al., 2004; Saleh et al., 2003). Soil, plant debris and seed from infected areas could potentially carry inoculum and introduce the pathogen to Australia. Soil and plant debris could potentially be carried on clothes, footwear, shipping containers, vehicles and machinery, while seeds could be imported legally or illegally. The entry potential of *H. maydis* can be considered as **High**.

5.1.9.2 ESTABLISHMENT POTENTIAL

Rating: High

H. maydis is present in a number of countries overseas (see Table 4) and parts of Australia are thought to have suitable climatic conditions for the pathogen (see Section 5.1.6). Host plants of *H. maydis* (i.e. maize, lupins and cotton) are also widespread. This pathogen could therefore become established in Australia, should it enter the country and come into contact with a suitable host plant. Therefore the establishment potential of *H. maydis* is considered as **High**.

5.1.9.3 SPREAD POTENTIAL

Rating: High

This pathogen could be spread by the movement of contaminated soil, plant debris and seed between properties adhering to machinery, clothing or footwear. The movement of maize silage between properties could potentially spread the pathogen, as could the movement of seed between properties. Therefore the spread potential of *H. maydis* can be considered as **High**.

5.1.9.4 ECONOMIC IMPACT

Rating: High

The pathogen has a significant impact on lupins (Bergstrom et al., 2008) however it is maize that is most severely affected. *H. maydis* has caused 40% yield losses in infected maize crops overseas (Zeller et al., 2000). Therefore it is likely that the pathogen would have a negative impact on Australian maize and lupin producers, depending on the pathotype involved in the incursion. Therefore the economic impact of *H. maydis* is considered to be **High**.

5.1.9.5 OVERALL RISK

Rating: High

Based on the individual ratings above, the combined overall risk of *H. maydis* is considered **High**.

5.2 Pest Details – Downy Mildew of Sunflower (*P. halstedii*)

Common name:	Downy Mildew of Sunflower
Scientific name:	<i>Plasmopara halstedii</i> (Farl.) Berl. & De Toni
Synonyms:	<i>Plasmopara helianthi</i>
Taxonomic position:	Kingdom: Chromista Phylum: Oomycota Class: Oomycetes Order: Peronosporales Family: Peronosporaceae Genus: <i>Plasmopara</i>

The information from this plan has been primarily obtained from documents as cited in the reference section as well as from a Pest Risk Review of Downy mildew (*Plasmopara halstedii*) (Plant Health Australia 2005).

5.2.1 Background

Downy mildew of sunflower was chosen as an example of a soil-borne pathogen that is recognised as having a high overall risk to the sunflower industry and as a High Priority Pest in the Grains Industry Biosecurity Plan (Plant Health Australia 2009, currently under review).

Downy mildew of sunflower (*Plasmopara halstedii*) is a pathogen found in most sunflower (*Helianthus annuus*) producing areas of the world; however Australia remains free of this disease. This pathogen was first discovered in Indiana, Iowa and Minnesota in the 1920s and is capable of causing significant yield losses overseas with losses of up to 39% reported in Iran (Roeckel-Drevet et al., 2003). However the damage caused by outbreaks of this pathogen is minimised by the use of resistant sunflower varieties (Zimmer 1974; Roeckel-Drevet et al., 2003).

Zimmer (1974) describes three formae speciales of this pathogen, *P. halstedii* f. sp. *helianthi*, *P. halsedii* f. sp. *perennis* and *P. halsedii* f. sp. *patens*. Of these *P. halstedii* f. sp. *helianthi* is the formae speciales that is a pathogen of sunflowers. There are currently 35 known races (pathotypes) of this pathogen that attack sunflowers (Sakr et al., 2008a).

P. halstedii has previously been recorded in Australia and New Zealand on cape weed, *Arctotheca calendula*, however subsequent investigations by Constantinescu and Thines (2010) have found that the pathogen is not *P. halstedii* but a new fungus described as *Plasmopara majewskii*.

5.2.2 Disease cycle

Oospores are produced in the roots of the host plant. As the crop breaks down (or is ploughed in) the sexual oospores enter the soil where they can remain viable for up to ten years (Vear et al., 2008).

The disease affects young seedlings when soil moisture is high and maximum temperature is between 15 and 18°C (Sakr et al., 2008b). When conditions are suitable and a susceptible host plant is available the oospores (the primary inoculum) germinate and produce zoosporangia, which in turn

produce motile zoospores (asexual spores) that infect the roots (and occasionally stems and leaves) of young sunflower plants. Once successfully established the infection then becomes systemic and spreads through the plant. Under favourable conditions the undersides of the leaves become covered in white zoosporangia. The zoosporangia can be blown short distances by the wind onto the leaves of other plants, spreading the infection over small distances. Sporangia can act as secondary inoculum. Oospores are simultaneously produced in the roots of the infected plants to act as an overwintering mechanism allowing the persistence of the fungus when host plants are not available.

There are two infection pathways utilised by this pathogen; oospores that are produced in the roots and zoosporangia produced on the leaves.

5.2.3 Dispersal

This pathogen can be dispersed in one of three ways.

- Oospores can be dispersed in soil and crop debris.
- Zoosporangia can be dispersed by the wind onto the leaves of other plants.
- It has also been suggested that the fungus can be spread by the movement of sporangia or oospores contaminating seeds from infected plants (loos et al., 2007). Meaning the movement of contaminated seed could also spread the pathogen to new areas.

5.2.4 Host range

Plasmopara halstedii has a wide host range including sunflowers and various other plants, however the forma specialis that affects sunflowers, *P. halstedii* f. sp. *helianthi*, has only been recorded in the field from; sunflower (*Helianthus annuus*) (Sakr et al., 2008b; Zimmer 1974), common ragweed (*Ambrosia artemisiifolia*) and common cocklebur (*Xanthium strumarium*) (Walcz et al., 2000), which all belong to the Asteraceae plant family.

Walcz et al., (2000) also describe a number of hosts that were able to be inoculated and infected by sunflower pathogenic pathotypes of *P. halstedii* under laboratory conditions (Table 5). These alternative hosts should be considered in surveys for this pathogen as research has shown that they are able to be infected, even if they have not been recorded as field hosts. *A. vulgaris* and *C. cyanus* are garden plants, highlighting the need to include gardens in any surveys for this pathogen.

Table 5 Alternative hosts of *P. halstedii* identified by Walcz et al., (2000) from inoculation experiments

Species name	Common name	Family
<i>Artemisia vulgaris</i>	Common wormwood	Asteraceae
<i>Centaurea cyanus</i>	Cornflower	Asteraceae
<i>Helianthus argophyllus</i>	Silver leaf sunflower	Asteraceae
<i>Helianthus debilis</i>	Beach sunflower	Asteraceae
<i>Helianthus divaricatus</i>	Woodland sunflower	Asteraceae
<i>Helianthus grosseserratus</i>	Sawtooth sunflower	Asteraceae
<i>Helianthus petiolaris</i>	Prairie sunflower	Asteraceae

5.2.5 Current geographic distribution

Downy mildew of sunflower has been recorded on all sunflower producing continents other than Australia. A list of the countries where the pathogen has been reported is given in Table 6.

Table 6 Geographic distribution of Downy mildew of sunflower

Country	Continent	References
Argentina	South America	Roeckel-Drevet et al., (2003); Gulya et al., (1991)
Bulgaria	South America	Roeckel-Drevet et al., (2003); Gulya et al., (1991)
Canada	North America	Roeckel-Drevet et al., (2003); Zimmer (1974); Gulya et al., (1991)
China	Asia	Roeckel-Drevet et al., (2003)
France	Europe	Roeckel-Drevet et al., (2003); Gulya et al., (1991)
Germany	Europe	Heller-Dohmen et al., (2011)
Hungary	Europe	Roeckel-Drevet et al., (2003); Gulya et al., (1991)
India	Asia	Agrawal et al., (1991)
Iran	Asia	Roeckel-Drevet et al., (2003)
Italy	Europe	Baldini et al., (2006)
Moldavia	Europe	Roeckel-Drevet et al., (2003)
Morocco	Africa	Roeckel-Drevet et al., (2003)
Romania	Europe	Roeckel-Drevet et al., (2003); Zimmer (1974)
Russia	Europe/Asia	Vear et al., (2008); Zimmer (1974); Roeckel-Drevet et al., (2003)
Serbia	Europe	Roeckel-Drevet et al., (2003)
South Africa	Africa	Roeckel-Drevet et al., (2003)
Spain	Europe	Molinero-Ruiz et al., (2002); Roeckel-Drevet et al., (2003)
Turkey	Asia/Europe	Evci et al., (2011)
Tunisia	Africa	Mounira et al., (2011)
Ukraine	Europe	Vear et al., (2008)
USA	North America	Molinero-Ruiz et al., (2002); Roeckel-Drevet et al., (2003); Zimmer (1974)
Yugoslavia	Europe	Zimmer (1974)

5.2.6 Potential geographic distribution in Australia

P. halstedii has a wide distribution (Table 6). The disease affects young seedlings when soil moisture is high and maximum temperature is between 15 and 18°C (Sakr et al., 2008b). The disease favours humid conditions for the infection of plant roots (Tourvieille de Lobrouhe et al., 2008). Areas with these environmental conditions and susceptible host plants include much of the current sunflower producing regions of Australia.

5.2.7 Symptoms

P. halstedii causes a range of symptoms. The most obvious symptoms are:

- Stunting (caused by a shortening of the distance between stem nodes) (see Figure 6)
- Discolouration of the upper surface of the leaf (Molinero-Ruiz et al., 2005; see Figure 7). The upper leaf surface discolouration corresponds with the presence of white zoosporangia on the underside of the leaves (another characteristic symptom of the pathogen)
- In cool (15-18°C), wet conditions damping off of young sunflower plants can be associated with this disease (Molinero-Ruiz et al., 2005).

On *A. artemisifolia* symptoms of leaf chlorosis and stunting were observed by Walcz et al., (2000). No symptoms were described for *P. halstedii* infections on *X. strumarium*.



Figure 6 Stunting symptoms on sunflower caused by a shortening of the distance between stem nodes in response to *P. halstedii* infection. Source: Ferenc Viranyi, Godollo University of Agricultural Sciences, bugwood.org



Figure 7 Leaf discoloration on the upper surface of the leaf caused by *P. halstedii*. Source: SRPV, Ile de France Archive, Les Services Régionaux de la Protection des Végétaux, bugwood.org

5.2.8 Diagnostic information

No National Diagnostic Protocol has been developed or endorsed for *P. halstedii*.

For diagnostic facilities and advisory services that can be utilised in the event of an incursion see Section 10.2 Appendix 2.

5.2.8.1 MORPHOLOGICAL AND PHYSIOLOGICAL DIAGNOSIS

The symptoms caused by the fungus on host plants (see Section 5.2.4) and the morphology of the fungus can aid in its identification.

Details on how to isolate oospores from host material is given in Spring and Zipper (2000) who studied the germination of *P. halstedii* oospores. They found that the oospores of *P. halstedii* are thick walled, round in shape and approximately 30 µm in diameter. After germination a germ tube of up to 750 µm (average germ tube length was approximately 100 µm) is produced giving rise to one or more zoosporangia.

Zoosporangia are usually either round or oval however some also produce pear shaped zoosporangia. Mean zoosporangia size is variable with sizes ranging from 377.3 to 543.3 µm² (Sakr et al., 2008b) with the zoosporangia size varying with the strain and the cultivar of host plant used in the experiment. Kulkarni et al., (2009) observed round zoosporangia of 12.5 µm in diameter and others that were oval and approximately 40 µm by 22.5 µm, with different pathotypes having different sized zoosporangia. Mounira et al., (2011) described the zoosporangia as being 20 to 25 µm by 13 to 19 µm.

Because of the natural variation in the morphology of *P. halstedii*, diagnosis using the symptoms and morphology of the fungus requires validation by molecular techniques such as PCR.

5.2.8.2 PCR

PCR has been successfully used by loos et al., (2007) to diagnose and detect *P. halstedii* in 35g sunflower seed samples. A PCR technique has also been developed by Roeckel-Drevet et al., (1999) to detect the pathogen in plant tissue samples.

5.2.9 Pest risk analysis – Downy mildew of sunflower

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

5.2.9.1 ENTRY POTENTIAL

Rating: High

Downy mildew of sunflower is found in most sunflower producing countries other than Australia. The pathogen can be spread by the movement of oospores in soil or crop debris, the wind-borne dispersal of zoosporangia between plants (Molinero-Ruiz et al., 2002) and by the movement of seed from infected plants (loos et al., 2007). The accidental movement of the pathogen’s oospores on soil and plant debris adhering to machinery, vehicles, footwear or clothing, and the importation of contaminated seed are potential ways this pathogen could enter Australia. Wind-borne dispersal of zoosporangia is more likely to be responsible for localised spread of the pathogen after it has entered the country. The entry potential of *P. halstedii* into Australia is considered to be **High**.

5.2.9.2 ESTABLISHMENT POTENTIAL

Rating: High

P. halstedii has a very wide distribution, occurring in most of the world’s sunflower growing countries. The pathogen is very adaptable and therefore could establish in Australia’s sunflower growing regions.

Oospores are known to remain viable in the soil for up to ten years (Vear et al., 2008) suggesting that should the pathogen enter the country it may be ten years before infected areas can be planted to host plants and be declared pest free again.

Host plants are found throughout Australia. Large parts of New South Wales and Queensland are planted to sunflower each year. Numerous sunflower varieties are planted as ornamental garden plants, and the alternative hosts’ *A. artemisifolia* and *X. strumarium* (see Section 5.2.4) are widespread weed species. The presence of widespread host plants increases the establishment potential of the pathogen. The establishment potential of this pathogen in Australia is considered to be **High**.

5.2.9.3 SPREAD POTENTIAL

Rating: High

Once in Australia spread could potentially occur:

- Through the movement of machinery, contaminated with soil or plant debris, between infected and non-infected paddocks and properties
- By the movement of contaminated seed between areas
- By the spread of wind-borne zoospores from the leaves of infected plants to non-infected plants. This is an important source of secondary infection and localised spread of the pathogen

Based on this information the potential for spread of *P. halstedii* following establishment is considered to be **High**.

5.2.9.4 ECONOMIC IMPACT

Rating: High

Roeckel-Drevet et al., (1999) describe the pathogen as one of the most serious diseases of sunflower, causing significant yield losses of up to 39% (Roeckel-Drevet et al., 2003). *P. halstedii* is particularly virulent during periods of frequent rainfall (Vear et al., 2008). Should an incursion occur there would also be additional costs to producers for fungicides and/or the acquisition of resistant sunflower cultivars, which are used to control the pathogen overseas (Vear et al., 2008; loos et al., 2007). The economic impact of this pathogen is expected to be **High**.

5.2.9.5 OVERALL RISK

Rating: High

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

6 Pest management

6.1 Response checklist

The following checklist (Table 7) provides a summary of generic requirements to be identified and implemented within a Response Plan for an incursion of a new soil-borne pathogen into Australia.

Table 7. Checklist of requirements to be identified in a Response Plan

Checklist item	Further information
Destruction methods for plant material, soil and disposable items	Section 7.1.1, 7.1.2
Disposal procedures	Section 7.1.5
Quarantine restrictions and movement controls	Section 7.3
Decontamination and property clean up procedures	Section 7.5
Diagnostic protocols and laboratories	Sections 5.1.8, 5.2.8 and 10.2
Trace back and trace forward procedures	Section 7.6
Protocols for delimiting, intensive and ongoing surveillance	Section 6.2
Zoning	Section 7.4
Reporting and communication strategy	Section 10.3

For 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).

6.2 Surveys and epidemiology studies

Information provided in Sections 6.2.1 to 6.2.3 provides a framework for the development of early detection and delimiting surveys for soil-borne pathogens.

As soil-borne pathogens are spread by the movement of inoculum in soil and plant debris it is important that personnel avoid moving infested soil between paddocks and properties. Footwear, tools, equipment and vehicles should be thoroughly washed of soil and plant debris and then sanitised with a registered disinfectant. Extra precautions should be taken when working areas known to be infested by the pathogen.

6.2.1 Technical information for planning surveys

When developing surveys for presence and/or distribution of the soil-borne pathogen, the following characteristics provide the basic biological knowledge that informs the survey strategy:

- Plant material may be asymptomatic, or may not display obvious symptoms at all growth stages (for example Late wilt of maize will often go undetected until the tasseling stage).
- Host species in Australia are likely to be numerous and widely dispersed and may be present within farm paddocks, as well as home gardens, landscape plantings, nurseries and as weeds.
- The risk of pathogen movement on plant material, machinery, equipment and personal effects is high.
- Some soil-borne pathogens can also be dispersed via seed or wind-borne inoculum (e.g. Downy mildew of sunflower).
- Production areas and significant proportions of Australia may have favourable climatic conditions for the pathogens spread and establishment.

6.2.2 Surveys for early detection of an incursion

Points to consider in effectively monitoring soil-borne pathogen populations are:

- Ensure that the laboratory diagnostician has expertise in this form of diagnosis.
- Initial surveys should concentrate on symptomatic plants (i.e. plants showing wilting, stunting or discoloration which can be associated with soil-borne pathogens).
- If pathogens are detected, or suspected, whole plant samples including roots should be collected for diagnosis.

Points to consider in monitoring infected material are:

- The host range of the potential pathogen incursion must be determined and hosts grouped into risk categories for 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. For example some pathogens are more likely to be visible during some climatic conditions or growth stages but not others, e.g. Late wilt of maize is often only apparent at the tasseling stage.
- Potential pathways for distribution of pathogen infected material must be determined.
- Depending on the pathogen, distribution of the pathogen in the plant may be irregular and plant material with most likely infection should be determined (e.g. are seed heads, leaves or roots most commonly infected by the pathogen).
- Depending on the pathogen, host species in Australia are likely to be numerous and widely dispersed and may be present within farms, nurseries, home gardens, landscape plantings, or as weeds.

- Mycologist 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 crops is essential to prevent the introduction of a soil-borne pathogen and limit its spread within and from contaminated areas. Where possible, early detection of disease symptoms while the pathogen is present at low levels, will provide the best chance of eradication success.
- Personnel involved in surveys must be trained to recognise particular symptoms of the pathogen and other similar disorders for comparison. For example if an incursion of Downy mildew of sunflower occurred personnel involved in the survey would need to be able to separate the symptoms of Downy mildew of sunflower (*P. halstedii*) from the endemic Sunflower powdery mildew (*Golovinomyces cichoracearum*).

6.2.3 Delimiting surveys in the event of an incursion

Delimiting surveys should comprise local surveys around the area of initial detection concentrating on areas of poor growth. The normal procedure is to collect symptomatic plants and to test them to confirm the presence of the pathogen. If confirmed, plants taken at random from the same crop should be tested to enable an estimate to be made of the disease incidence. Surrounding crops would then be surveyed. The extent of the survey beyond the initial infected crop should be guided by the test results from surrounding crops.

If a pathogen can be seed transmitted seed trace-back 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.

In the event of an incursion, delimiting surveys are essential to 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, as well as distribution pathways for soil and plant material and potentially the weather patterns experienced during the period prior to detection (Figure 8). Other considerations are, for example, the movement of people, plant material or equipment as a result of trace-forward and trace-backs.
- Soil-borne pathogens can be spread between areas by the movement of soil and plant material adhering to machinery, vehicles, livestock or people.
- Some soil-borne pathogens can be spread by the movement of seed.
- All potential host species of the pathogen (see Sections 5.1.4 and 5.2.4) should be surveyed, with particular attention paid to the species in which the pathogen was initially detected.
- In addition to inspection of possible host plants, material should be collected for diagnostic purposes (refer to Section 6.2.4).
- If the incursion is in a populated area, publication and distribution of information sheets and appeals for public assistance may be helpful.

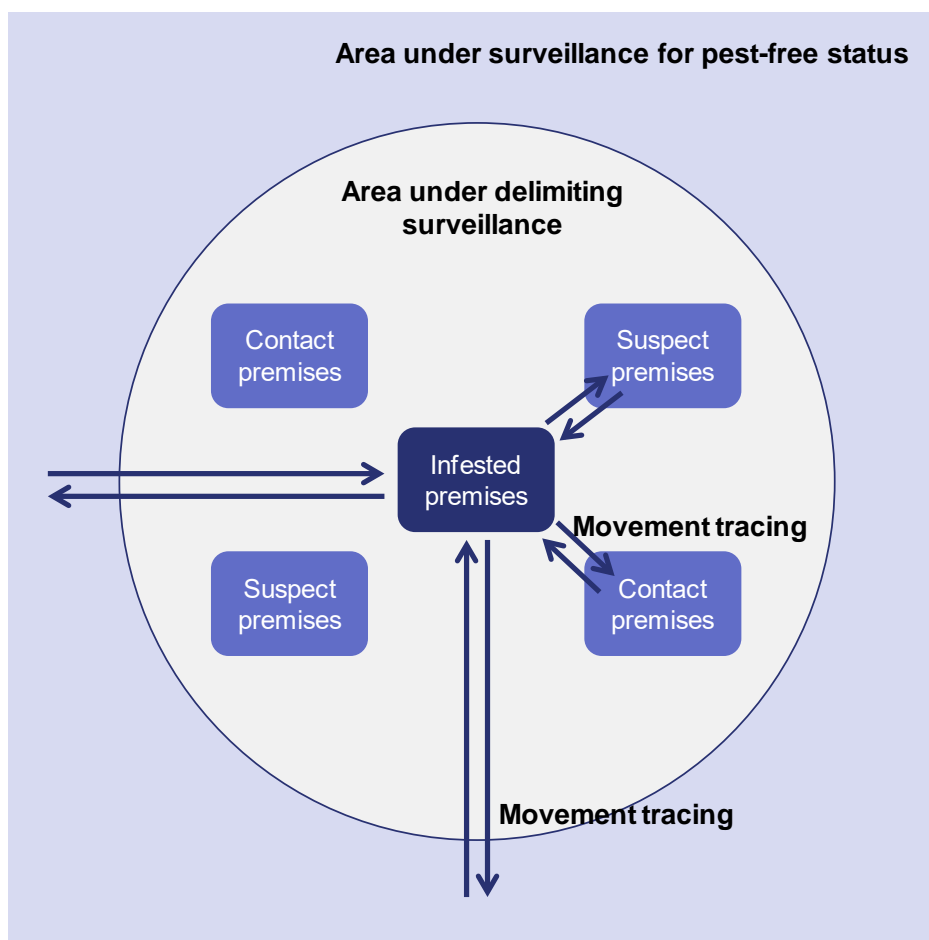


Figure 8. Diagram of a delimiting survey showing surveillance activities from the infested premises

6.2.4 Collection and treatment of samples

Once initial samples have been received and preliminary diagnosis made, follow up samples to confirm identification of the pathogen will be necessary. This will involve sampling directly from the infested crop, and sampling crops over a larger area to determine the extent of disease distribution.

From each crop sampled, samples from at least 100 plants should be taken at random (the exact number will depend on 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 5.1.7 and 5.2.7 for full details.

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 at this point 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. Follow up samples will be forwarded to the nominated diagnostic laboratories for processing.

Samples should be initially collected over a representative area of the infected crop to determine the pathogen's distribution. The disease may appear as patches within the crop depending on the source of the pathogen.

It is important to note the distribution of disease in the initial infected crop, as this will indicate whether the pathogen has been seed-borne, carried on trash 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 or human health impacts 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. In addition containers should be clearly labelled in accordance with the requirements of PLANTPLAN (Plant Health Australia 2013). 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.

Refer to PLANTPLAN for packing instructions.

6.2.4.1 HOW TO TREAT SAMPLES

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. Samples should be wrapped in damp newspaper, bundled into a plastic bag and clearly labelled. 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).

Samples should be processed as quickly as possible after sampling from the field if sub-cultures are to be made from infected tissue. Once removed from the field, fresh plant samples can deteriorate and become contaminated by other mould, fungi and bacteria, which may prevent successful sub-culturing of the pathogen. Sub-culturing should be done within three to four days after sampling from the field.

Infected plant tissue to be used for PCR analysis can be placed in a -80°C freezer and stored for an indefinite period without damaging fungal DNA.

Long term storage of fungal isolates can occur and be freeze dried for future reference (without loss of viability) or as deep frozen plant specimens maintained at -80°C, which can be used to extract DNA.

6.2.5 Epidemiological study

There are many factors that affect the development of soil-borne pathogens in fields. These include: the presence of virulent strains in the soil, susceptibility of the crop varieties, soil type, soil fertility, climatic conditions, irrigated or non-irrigated crops and interactions with other soil-borne micro-organisms. Inoculum densities in the soil are also important as disease symptoms may not be apparent when there are low levels of the pathogenic strains in the soil.

The number of infected plants within a crop will depend on the source and amount of primary inoculum available and whether environmental conditions have been favourable for the pathogen to spread from initial foci.

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 infestation source, including both 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, vegetable, fodder, pasture and garden plants.
- Machinery or vehicles that have been into the infested area or in close proximity to the infestation source. This is especially important due to the possible movement of contaminated soil on machinery moving between areas.
- The extent of human movements into and around the infested area. A possible link to the recent overseas travel or visitors from other regions or the recent importation of plant material, machinery or goods that could contain soil from other regions should also be considered.
- The source of seed and how long that seed has been used by the grower, this is especially important if the pathogen could be dispersed with seed. If any other crops have been sown from the same source seed.
- The temperature and environmental conditions. Temperature and environmental conditions affect the severity and spread of the pathogen and therefore need to be considered.

6.2.6 Models of spread potential

6.2.6.1 *HARPOPHORA MAYDIS* – LATE WILT OF MAIZE

No models were found that looked at the spread of this pathogen. Future models would need to consider how the pathogen is dispersed (see Section 5.1.3), the pathogen's host range (see Section 5.1.4), host availability and its climatic requirements.

6.2.6.2 *PLASMOPARA HALSTEDII* – DOWNY MILDEW OF SUNFLOWER

No models of spread potential have been developed for *P. halstedii*. If models were to be developed they would need to consider how the pathogen is dispersed (see Section 5.2.3), the pathogens host range (see Section 5.2.4), host availability and the pathogens climatic requirements.

6.2.7 Pest Free Area guidelines

The establishment and maintenance of pest free areas (PFAs) would be 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.

Determination of Pest Free Areas (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 6.2 for points to consider in the design).
- Plant sampling should be based on at least 100 plants taken at random per crop.
- Preliminary diagnosis can be based on plant symptoms and fungal morphology.
- PCR methods for confirmation of fungal identity.
- Seed sampling (if the pathogen 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.
- Surveys should also consider alternative host plants (see Sections 5.1.4 and 5.2.4) and not be limited to the primary infected host.
- Information (including absence of the pest) should be recorded.

6.3 Availability of control methods

Once introduced and established, many soil-borne pathogens can survive in soil for extended periods, even in the absence of crop hosts, making eradication a long term process. For example *P. halstedii* oospores are known to remain viable in the soil for up to ten years (Vear et al., 2008) and therefore it may take ten years before an infected area can be declared pest free (the time required depends on the survival ability of the pathogen's inoculum). Containment procedures to retard the spread of the pathogen are required to minimise the impact on the industry and improve the probability of eradication success.

The following outlines some general information for the control of exotic soil-borne pathogens.

6.3.1 General procedures for control

Control of soil-borne pathogens is likely to be largely reliant on the use of fungicides and reducing the spread of the pathogen between areas by controlling the movement of people, machinery and equipment. Specific control measures will be determined by a CCEPP, however, general procedures include:

- Keep traffic out of affected areas and minimise movement in adjacent areas.
- Adopt best-practice property hygiene procedures to retard the spread of the pest between paddocks 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, including seed if the pathogen can be seed-borne, is an effective control.
- Avoid including host plants in crop rotations, for example if *P. halstedii* was found sunflowers and other host plants should be removed from the crop rotation (including volunteer plants and weeds).
- On-going surveillance of infected areas to ensure the pest is eradicated. As many soil-borne pathogens can survive for many years without host plants surveillance may have to be maintained for many years. The exact time will be determined by the inoculum's ability to survive in the absence of host plants. For example *P. halstedii* can survive in the soil for up to ten years (Vear et al., 2008).
- Do not use seed from infected plants for sowing, as seeds can be a source of inoculum for some soil-borne pathogens (e.g. *H. maydis* (Johal et al., 2004; Saleh et al., 2003) and *P. halstedii* (loos et al., 2007)).

6.3.2 Control of infected areas

6.3.2.1 CONTROL IF SMALL AREAS ARE AFFECTED

If a small area is found to be infected pull out the affected plants, as well as healthy plants 5-10 metres into the area surrounding the infected patch. Burn all plants in the affected patch. Particular care must be taken to minimize the transfer of infected soil from the area. Raking and burning the whole field at this stage is NOT an option as this procedure is likely to spread the pathogen over the field.

Host plants should not be planted in the infected paddock for several years; the exact time will depend on the soil life of the inoculum.

All equipment, including footwear, 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 property. The clean down procedure should be carried out on a hard surface or preferably a designated wash-down area to avoid mud being recollected from the affected site onto the machine.

6.3.2.2 CONTROL IF LARGE AREAS ARE AFFECTED

If a large area is infected, kill any surviving plants in the area, preferably with herbicides (note herbicides have to be registered by the Australian Pesticides and Veterinary Medicines Authority (APVMA) for the purpose), treat the debris with an appropriate fungicide and burn the crop debris (if it is thought that viable spores will not be spread by the updraft created by the fire). Once the dead plants have broken down, plant an alternative non-host crop or pasture to prevent erosion.

Particular care must be taken to minimise the transfer of inoculum, plant material or soil from the area and surveys of the surrounding area must continue for some time to ensure that the eradication regime was successful. The length of time that surveys continue will be determined by the survival ability of the pathogen's inoculum.

All equipment used on the site should be thoroughly cleaned down (as per section 6.3.2.1).

6.3.3 Weed management

Weeds can serve as alternate hosts of many pathogens. For example *A. artemisifolia* and *X. strumarium* are alternative hosts of *P. halstedii* (Walcz et al., 2000) and are considered to be weeds in Australia. If weed species are found to be potential hosts of the pathogen they will also 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.

6.3.4 Chemical control

Fungicidal chemicals can be applied as foliar sprays, seed dressings, or applied in a granular form at sowing. Unlike seed dressings foliar sprays are able to be applied to the growing crop. However many are unsuitable for controlling soil-borne pathogens in growing crops as it is difficult to get the chemical in contact with the pathogen. Destruction of the hosts, using herbicides, may be a more appropriate method of eradicating the pathogen when detected in a living crop as this will stop the pathogens lifecycle and limit the amount of inoculum that it produces.

Fungicidal seed dressings are often used to minimise the negative impacts of soil-borne pathogens and may be useful as preventative measures on properties surrounding the destruction zone, helping reduce the spread of the pathogen.

The exact chemicals that could be used for the control/eradication of a soil-borne pathogen will need to be considered on a case by case basis as the chemicals used and rates that they are applied can vary greatly between pathogens and crops.

Table 8 details the chemicals that have been used to control *H. maydis* and *P. halstedii* overseas. However any chemical used in Australia as part of a control or eradication program must be approved for that use by the APVMA before it can be used in Australia.

Table 8. Chemicals used overseas to control *H. maydis* and *P. halstedii*

Chemical	Mode of application in reference	Reference	Is the chemical registered in Australia?
Late wilt of maize (<i>Harpophora maydis</i>)			
Benomyl	Soil drench	Singh and Siradhana (1989)	No
Carbendazim ¹¹	Soil drench	Singh and Siradhana (1989)	Yes
Triadimefon	Soil drench	Singh and Siradhana (1989)	Yes
Downy mildew of sunflower (<i>Plasmopara halstedii</i>)			
Azoxystrobin ¹²	Effective as both a seed dressing and foliar spray but most effective when applied as seed dressing followed by a foliar spray soon after seedling emergence.	Sudisha et al., (2010)	Yes
Kresoxim-methyl	Effective as both a seed dressing and foliar spray but most effective when applied as seed dressing followed by a foliar spray soon after seedling emergence.	Sudisha et al., (2010)	Yes
Mefenoxam ¹³	Seed dressing	Molinero-Ruiz et al., (2005)	No
Metalaxyl ¹⁴	Seed dressing	Molinero-Ruiz et al., (2005)	Yes
Pyraclostrobin ¹⁵	Foliar spray. Suitable for use on a range of plants to control various fungal pathogens.	Daughtrey and Benson (2005)	Yes
Trifloxystrobin	Effective as both a seed dressing and foliar spray but most effective when applied as seed dressing followed by a foliar spray soon after seedling emergence.	Sudisha et al., (2010)	Yes

¹¹ An APVMA review is in progress for this chemical (see www.apvma.gov.au).

¹² A seed treatment containing this chemical is currently registered under the trade name "Dynasty[®]" in the USA to control *P. halstedii*

¹³ No information available on this chemical on the APVMA website (see www.apvma.gov.au)

¹⁴ Note: Molinero-Ruiz et al., (2005) suggests that resistance to this chemical is beginning to develop in Europe. Suggesting that the susceptibility of pathotype involved in the incursion to this chemical should be determined before widely using this fungicide to control *P. halstedii* incursions in Australia.

¹⁵ Chemical is registered in the USA (e.g. under the trade name "Headline[®]" to control this pathogen on sunflowers.

6.3.5 Cultural Control

Due to the long survival life of fungal spores in the soil few cultural controls are used to control soil-borne pathogens. Crop rotations that do not include susceptible host plants are not always a viable option, except during eradication efforts or if the pathogen's inoculum is relatively short lived (as is the case with *H. maydis* which has a soil life of only 12 to 15 months (USDA 2011; Sabet et al., 1970), and can therefore be managed using crop rotations). If using crop rotations to control the pathogen care should be taken to control weeds and volunteer plants as these could also act as a source of disease if not controlled.

The use of disease free seed is a simple cultural control practice that can be used to minimise the risk of introducing pests and diseases including soil-borne pathogens. The use of resistant host plants can also be a simple way of managing many soil-borne pathogens.

6.3.6 Host-Plant Resistance

The development of resistant host plants offers a low cost way of managing soil-borne pathogens. For example *P. halstedii* is often controlled overseas by using resistant sunflower varieties (e.g.: Jovic et al., 2010; Vear et al., 2008; Vear et al., 1997) and *H. maydis* can be controlled by the use of resistant maize varieties (e.g. Mosa et al., 2010).

6.3.7 Biological control

The use of biological control of soil-borne pathogens is not common; however there are some options available for the control of Late wilt of maize and Downy mildew of sunflower.

Some antagonistic fungi and bacteria have been identified as being useful to biologically control *H. maydis* (Table 9). No biological controls have been used to control *P. halstedii*, although a virus has been isolated from *P. halstedii* infecting sunflowers (Heller-Dohmen et al., 2011) suggesting that there may be some organisms that are antagonistic towards this fungus. However these have not yet been utilised for the biological control of the pathogen.

Table 9 Fungi found to be antagonistic towards *H. maydis*.

Biological control	Life form of biological control agent	Reference
<i>Candida glabrata</i>	Fungus	El-Mehalawy et al., (2004)
<i>Candida maltosa</i>	Fungus	El-Mehalawy et al., (2004)
<i>Candida slooffii</i>	Fungus	El-Mehalawy et al., (2004)
<i>Rhodotorula rubra</i>	Fungus	El-Mehalawy et al., (2004)
<i>Streptomyces graminofaciens</i>	Fungus	El-Mehalawy et al., (2004)
<i>Streptomyces gibsonii</i>	Fungus	El-Mehalawy et al., (2004)
<i>Streptomyces lydicus</i>	Fungus	El-Mehalawy et al., (2004)
<i>Streptomyces nogalater</i>	Fungus	El-Mehalawy et al., (2004)

Biological control	Life form of biological control agent	Reference
<i>Streptomyces rochei</i>	Fungus	El-Mehalawy et al., (2004)
<i>Streptomyces annulatus</i>	Fungus	El-Mehalawy et al., (2004)
<i>Trichosporon cutaneum</i>	Fungus	El-Mehalawy et al., (2004)
<i>Trichurus spiralis</i>	Fungi	Abd-El-Hamid et al., (1981)
<i>Bacillus subtilis</i>	Bacteria	Sellam et al., (1978)

7 Course of action – eradication methods

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 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.

7.1 Destruction strategy

7.1.1 Destruction protocols

- Infected crops should be destroyed by the application of an herbicide (and fungicide if a suitable chemical is available for controlling the pathogen in question) followed by the destruction of the crop by burning (if it is determined that the updraft will not spread viable spores to new areas). Care must be taken to minimise the spread of soil and plant debris from the site during this process. For this reason ploughing of crop debris may not be recommended.
- Disposable equipment, infected plant material or soil should be disposed of by autoclaving, high temperature incineration or deep burial.
- Any equipment removed from the site for disposal should be double-bagged.
- 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.

7.1.2 Decontamination protocols

If decontamination procedures are required, machinery, equipment and vehicles in contact with infected plant material or soil or present 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. use 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 soil-borne pathogen, additional or modified procedures may be required for the destruction of the pathogen. Any sterilisation procedure must be approved for use in the endorsed Response Plan.

7.1.3 Priorities

- Confirm the presence of the pathogen.
- Limit movement of people and prevent movement of vehicles and equipment through affected areas.
- Stop the movement of any plant material, soil or machinery that could be carrying fungal spores from the infected area.
- Determine the strategy for the eradication/decontamination of infected host material.
- Determine the extent of the 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 pathogen, this is especially important for seed transmitted pathogens such as Downy mildew of sunflower and Late wilt of maize.

7.1.4 Plants, by-products and waste processing

- Seeds harvested from infected plants and any soil or infected plant material removed from the infected site should be destroyed by (enclosed) high temperature incineration, autoclaving or deep burial (in a non-cropping area).
- Infested paddocks should remain free of susceptible host plants (including weeds, alternative hosts and volunteer plants) (see Sections 5.1.4, and 5.2.4) until the area has been shown to be free from the pathogen. In the case of some soil-borne pathogens this may take up to ten years as the inoculum of some species is able to survive in the soil for a long period of time. The exact period of time that the infested area should remain free of host plants will be determined by the survival ability of the inoculum of the pathogen in question.

7.1.5 Disposal issues

- Particular care must be taken to minimise the transfer of infected plant material and soil from the infected area.
- If the pathogen is also 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).
- Chemical resistance to Metalaxyl is developing in *P. halstedii* in Europe (Molinero-Ruiz et al., 2005) and would need to be considered if Metalaxyl is to be used for the control/eradication of *P. halstedii*.

7.2 Containment strategies

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

7.3 Quarantine and movement controls

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

7.3.1 Quarantine priorities

- Plant material (including seed if the pathogen is known to be seed-borne, as is the case for Late wilt of maize and Downy mildew of sunflower) and soil at the site of infestation to be subject to movement restrictions.
- Machinery, equipment, vehicles and disposable equipment in contact with infested plant material or soil, or present in close proximity to the site of infestation to be subject to movement restrictions.
- Harvesting of infected crops should be prevented as harvesting can, in some instances (e.g. Bunt infected crops), spread the disease to neighbouring areas.

7.3.2 Movement controls

If Restricted or Quarantine Areas are practical, movement of equipment or machinery should be restricted and movement into the area only occurs by permit. The industry affected will need to be informed of the location and extent of the disease occurrence.

Movement of people, vehicles and machinery, from and to affected farms, must be controlled to ensure that infected soil or plant debris (including seed) is not moved off-farm on clothing, footwear, vehicles or machinery. 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 double-bagged prior to removal for decontamination or should not leave the farm until thoroughly disinfected, washed and cleaned.
- Residents should be advised on measures to minimise the inadvertent transport of fungal spores from the infested area to unaffected areas.
- Plant material or plant products, including seed, must not be removed from the site unless part of an approved disposal procedure.
- All machinery and equipment should be thoroughly cleaned down with a high pressure cleaner (see Section 7.1.2) or scrubbing with products such as a farm degreaser or a 1% bleach (available chlorine) solution, prior to leaving the affected area. Machinery should be inspected for the presence of soil and plant debris and if found must be treated in an appropriate manner. The clean down procedure should be carried out on a hard surface, preferably a designated wash-down area, to avoid mud being re-collected from the affected site onto the machine. When using high pressure water, care should be taken to contain all plant material and mud dislodged during the cleaning process.
- Seed from the affected site should not be used for planting new crops, feeding stock or for human consumption. Hay, stubble or trash should not be removed from the site, as these materials could inadvertently spread the pathogen.

7.4 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 National Management Group during the production 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 9.

7.4.1 Establishing Quarantine Zones

Delimiting surveillance will inform the establishment of quarantine zones and identify the Restricted Area(s) (RA), Control Area (CA) and Pest Free Area (PFA). The size of each quarantine zone will be determined by a number of factors including location of the incursion, climatic conditions, pest biology and proximity of an Infected Premises (IP) to other IPs.

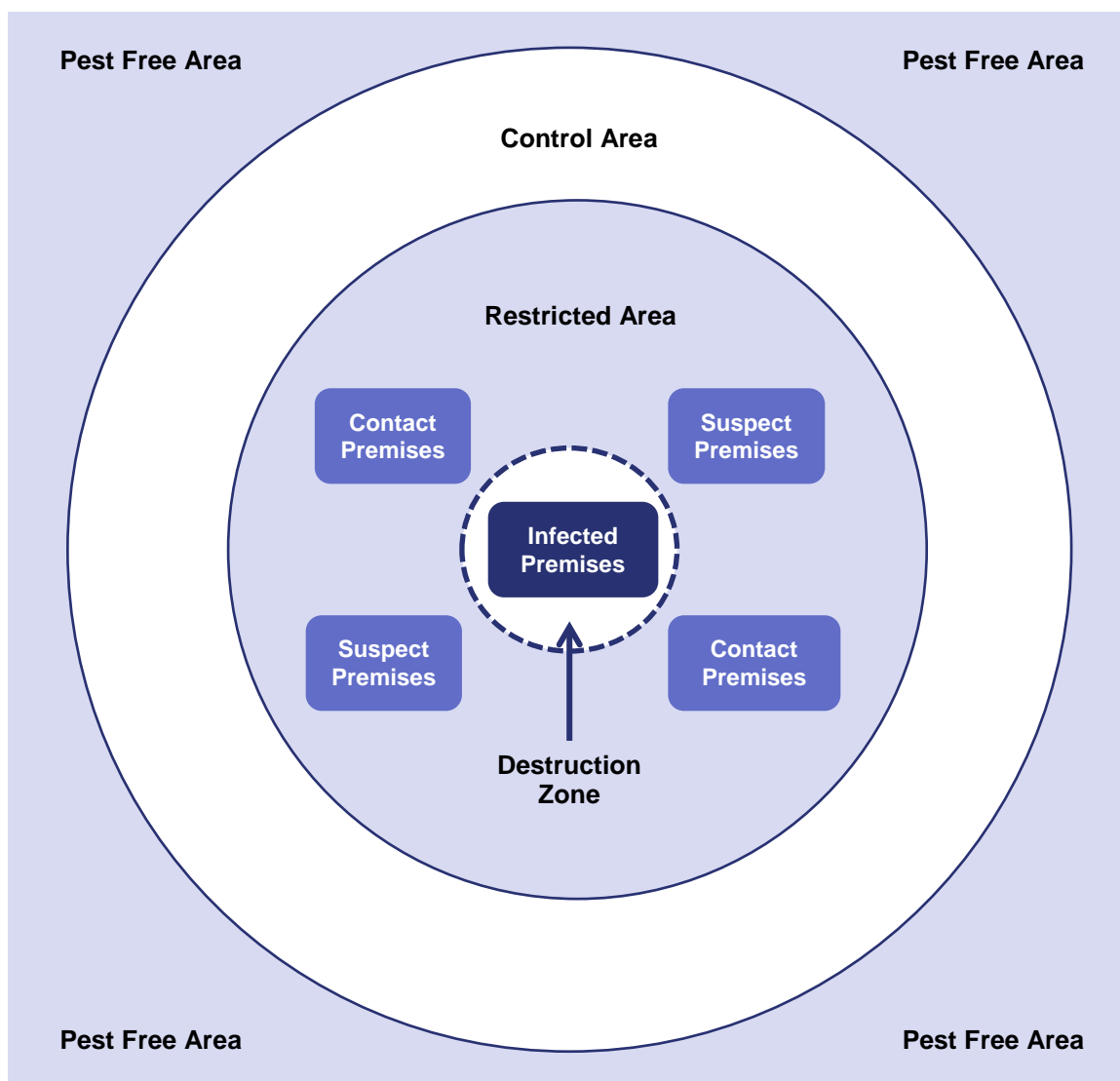


Figure 9. Schematic diagram of quarantine zones used during an EPP incursion (not drawn to scale)

7.4.2 Destruction Zone

The size of the Destruction Zone (i.e. zone in which the pest and all host material is destroyed) will depend on, distribution of the pest (as determined by delimiting surveys), ability of the pest to spread, factors which may contribute to the pest spreading and the time of season.

All host plants should be destroyed after the level of infection has been established. The delimiting survey will determine whether or not neighbouring host crops are infected and need to be destroyed. If spread is likely to have occurred prior to detection, the Destruction Zone may include contiguous areas that have been in contact with, or are associated with the same management practices as, the infected area. Particular care needs to be taken to ensure that plant material and soil are not moved into surrounding areas that are not showing symptoms of the pest. Where possible, destruction should take place in dry conditions to limit mud being spread within the field on boots and protective clothing.

7.4.3 Restricted Area

Data collected from surveys and tracing (trace back and trace forward) will be used to define the RA, which comprises all properties where the pest has been confirmed (Infected Premises or IP), properties which have come into direct or indirect contact with an IP or infected plants (Contact Premises or CP) and properties which may have been exposed to the pest (Suspect Premises or SP). The RA will be subject to intense surveillance and movement control, with movement out of the RA to be prohibited and movement into the RA to occur by permit only.

7.4.4 Control Area

A CA is established around a RA to control the movement of susceptible hosts and other regulated materials until the extent of the incursion is determined. There may be multiple RAs within one CA. When the extent of the EPP Incident has been confidently defined, the RA and CA boundaries and movement controls may need to be modified, and where possible reduced in size commensurate with appropriate controls.

Additional zones can be utilised as required for operational purposes.

7.5 Decontamination and farm clean up

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

7.5.1 Decontamination procedures

General guidelines for decontamination and clean up:

- Keep traffic out of affected area and minimise it in adjacent areas.
- Adopt best-practice property hygiene procedures to retard the spread of the soil-borne pathogen 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 7.1.2.
- Only recommended materials are to be used when conducting decontamination procedures, and must be applied according to the product label.

- Infested plant material should be disposed of by autoclaving, high temperature (enclosed) incineration or deep burial.

For further information, 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).

7.5.2 General safety precautions

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

7.6 Surveillance and tracing

7.6.1 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 pathogen.
- Surveying other host growing properties and backyards.

7.6.2 Survey regions

Establish survey regions around the surveillance priorities identified above. These regions will be generated based on the zoning requirements (see Section 7.4), and prioritised based on their potential likelihood to currently have or receive an incursion of this pest. Surveillance activities within these regions will either allow for the area to be declared pest free and maintain market access requirements or establish the impact and spread of the incursion to allow for effective control and containment measures to be carried out. Detailed information regarding surveys for soil-borne pathogen infected plant material have been outlined elsewhere in this plan (refer to Section 6.2).

Steps outlined in Table 10 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 10. Phases to be covered in a survey plan

Phase 1	<ul style="list-style-type: none"> 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	<ul style="list-style-type: none"> Preliminary survey of host crops on properties in buffer zone establishing points of pest detection.
Phase 3	<ul style="list-style-type: none"> Surveillance of an intensive nature, to support control and containment activities around points of pest detection.
Phase 4	<ul style="list-style-type: none"> 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 pathogen. Pathways to be considered are: <ul style="list-style-type: none"> Items of equipment and machinery which have been shared between properties including bins, containers, irrigation lines, vehicles and equipment. The producer and retailer of infected material if this is suspected to be the source of the outbreak. 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). Movement of plant material and soil from controlled and restricted areas. Storm and rain events and the direction of prevailing winds that result in air-borne dispersal of the pathogen during these weather events.
Phase 5	<ul style="list-style-type: none"> Surveillance of farms, gardens and public land where plants known to be hosts of the soil-borne pathogen are being grown.
Phase 6	<ul style="list-style-type: none"> Agreed area freedom maintenance, post-control and containment.

7.6.3 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 the eradication of soil-borne pathogens may include:

- Establishment of sentinel plants at the site of infection.
- Maintain good sanitation and hygiene practices throughout the year.
- Monitoring of sentinel plants for signs of the pathogen.
- Sentinel plants should remain in place and inspected on a fortnightly basis for a further 6 weeks and then on a monthly basis.
- If symptoms or pathogen are detected, samples are to be collected and stored and plants destroyed.
- Surveys comprising of host plant sampling for the pathogen should be undertaken for a defined period after eradication has been achieved (or as endorsed by a CCEPP). Note the biology of the pathogen will dictate the minimum number of years that surveys need to be undertaken for.

8 Technical debrief and analysis for stand down

Refer to section 4.3 of 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.

9 References

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10 Appendices

10.1 Appendix 1: 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).

10.2 Appendix 2: 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

10.3 Appendix 3: Communications strategy

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

10.4 Appendix 4: Market access impacts

Within the Department of Agriculture, Fisheries and Forestry (DAFF) Manual of Importing Country Requirements (MCoR) database (<http://www.daff.gov.au/micor/plants>) export of some material may require an additional declaration regarding freedom from the pathogen. Latest information can be found within MCoR, using a search for the particular pathogen (note as many fungal pathogens have scientific names for both their teleomorph (sexual) and anamorph (asexual) life stages both names should be searched for in the database).

The DAFF MCoR database was searched in August 2013 for current trade restrictions relating to the two soil-borne pathogens used as examples in this contingency plan. The findings of the search are summarised in Table 11.

Table 11 Countries identified on the DAFF MCoR database that have trade restrictions regarding *H. maydis* and/or *P. halstedii*

Country	Commodity	Requirements/restrictions
Late wilt (<i>Harpophora maydis</i> (syn. <i>Cephalosporium maydis</i>))		
New Zealand	<i>Zea mays</i> Maize – grains/seeds - sowing	An import permit is not required; however a phytosanitary certificate and declaration that Late wilt is not known to occur in Australia is required.
Sri Lanka	<i>Zea mays</i> Maize – grain/seeds – consumption	An import permit, phytosanitary certificate and declaration that Late wilt is not known to occur in Australia is required.
South Africa	<i>Zea mays</i> Maize – grain/seeds – research	An import permit, phytosanitary certificate and declaration that Late wilt is not known to occur in Australia is required.
South Africa	<i>Zea mays</i> Maize – grains/seeds - sowing	An import permit, phytosanitary certificate and declaration that Late wilt is not known to occur in Australia is required.
Tonga	<i>Zea mays</i> Maize/corn – grains/seeds - sowing	An import permit, phytosanitary certificate and declaration that Late wilt is not known to occur in Australia is required.
Downy mildew of sunflower (<i>Plasmopara halstedii</i>)		
European Union (EU)	<i>Helianthus annuum</i> Sunflower seed – grains/seeds – sowing	An import permit is not required; however a phytosanitary certificate and declaration that <i>P. halstedii</i> is not known to occur in Australia is required.
India	<i>Helianthus annuus</i> Sunflower seed – grains/seeds – research	An import permit, phytosanitary certificate and declaration that <i>P. halstedii</i> is not known to occur in Australia is required.
Iran	<i>Helianthus annuus</i> Sunflower seed – grain/seeds - sowing	An import permit is not required, however a phytosanitary certificate and declaration that <i>P. halstedii</i> is not known to occur in Australia is required.
New Caledonia	<i>Helianthus</i> spp. Sunflower seed – grain/seeds – sowing	An import permit, phytosanitary certificate and declaration that <i>P. halstedii</i> is not known to occur in Australia is required.
New Caledonia	Stockfeed	An import permit, phytosanitary certificate and declaration that <i>P. halstedii</i> is not known to occur in Australia is required.
New Zealand	<i>Helianthus</i> spp. Sunflower seed – grains/seeds - sowing	An import permit is not required; however a phytosanitary certificate and declaration that <i>P. halstedii</i> is not known to occur in Australia is required.
Papua New Guinea	<i>Helianthus</i> spp. Sunflower seed – grains/seeds sowing	An import permit, phytosanitary certificate and declaration that <i>P. halstedii</i> is not known to occur in Australia is required.
Serbia	<i>Helianthus annuus</i> Sunflower seed – grains/seeds – sowing	An import permit, phytosanitary certificate and declaration that <i>P. halstedii</i> is not known to occur in Australia is required.

Country	Commodity	Requirements/restrictions
Serbia	<i>Helianthus annuus</i> Sunflower seed – grains/seeds – consumption	An import permit, phytosanitary certificate and declaration that <i>P. halstedii</i> is not known to occur in Australia is required.
Switzerland	<i>Helianthus annuus</i> Sunflower seed – grains/seeds – sowing	An import permit is not required; however a phytosanitary certificate and declaration that <i>P. halstedii</i> is not known to occur in Australia is required.
Switzerland	<i>Helianthus annuus</i> Sunflower seed – grains/seeds – consumption	An import permit is not required; however a phytosanitary certificate and declaration that <i>P. halstedii</i> is not known to occur in Australia is required.
Tanzania	<i>Helianthus</i> spp. Sunflower seed – grains/seeds – sowing	An import permit, phytosanitary certificate and declaration that <i>P. halstedii</i> is not known to occur in Australia is required.
Turkey	<i>Helianthus</i> spp. Sunflower seed – grains/seeds – sowing	An import permit is not required; however a phytosanitary certificate and declaration that <i>P. halstedii</i> is not known to occur in Australia is required.