Australian Government

MARINE PESTS

Rapid response manual for *Undaria pinnatifida*

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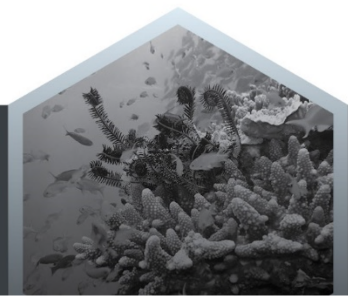

Nationally agreed guidance material endorsed by the Marine Pest Sectoral Committee

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Disclaimer

These manuals are part of a series of documents providing detailed information and guidance for emergency response to key marine pest species or groups of pest species.

The manuals are made available on the understanding that the Commonwealth of Australia is not thereby engaged in rendering professional advice. The Commonwealth does not warrant the accuracy, currency or completeness of the guidelines, or their relevance for any particular purpose. In particular, it should be noted that legislation, regulations and by-laws may vary between different jurisdictions and ports in Australia. Consequently, the guidelines do not purport to state what is necessary or sufficient to comply with laws applying in any place.

Before relying on the manuals in any important matter, users should obtain appropriate professional advice to evaluate their accuracy, currency, completeness and relevance for their purposes.

Note

Rapid response manuals are a key element of the Australian Emergency Marine Pest Plan. They provide detailed information and guidance for emergency response to a marine pest incident. The guidance is based on sound analysis and links policy, strategies, implementation, coordination and emergency management plans.

Preface

The Australian Government Department of Agriculture maintains a series of emergency response[1] documents to ensure national coordination of emergency responses to incursions by exotic pests and diseases or significant range expansions of established pests and endemic diseases. The Emergency Marine Pest Plan (EMPPlan) Rapid Response Manuals for marine pests provide detailed information and guidance for emergency response to key marine pest species or groups of pest species of national significance.

The EMPPlan is adapted from the Australian emergency plans for terrestrial and aquatic animal diseases—the Australian Veterinary Emergency Plan (AUSVETPLAN) and the Australian Aquatic Veterinary Emergency Plan (AQUAVETPLAN). The format and content have been kept as similar as possible to those documents to enable emergency response personnel trained in their use to work efficiently with these manuals in the event of a marine pest emergency.

This manual describes the principles for an emergency response to an incident caused by the suspicion or confirmation of incursion by the Japanese seaweed *Undaria pinnatifida*, a known invasive marine pest species established in Victoria and Tasmania but not considered to be widespread. It is listed on Australian Priority Marine Pest List.

Dr Graeme Inglis and Ms Kimberley Seaward from the National Institute of Water and Atmospheric Sciences, New Zealand, and Ms Amy Lewis from the Department of Agriculture prepared the first edition of this Rapid Response Manual. The manual was revised as part of activity 3.5 of MarinePestPlan 2018-2023 (plan and implement procedures to develop and update the EMPPLlan rapid response manuals and related guidance materials). The Marine Pest Sectoral Committee endorsed this manual.

The manual will be reviewed at least every five years to incorporate new information and experience gained with incursion management of these or similar marine pests. Amended versions will be published on the marine pest website.

[1] Note that the term 'emergency response' as used in this document does not refer to a 'biosecurity emergency' as that term is used under the *Biosecurity Act 2015*, nor do any activities described by this document undertaken during an 'emergency response' intended to be an exercise of powers provided by Chapter 8 (Biosecurity Emergencies and Human Biosecurity Emergencies) of that Act.

Recommendations for amendments

To recommend changes to this document, forward your suggestions to:

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Introduction

Emergency response operations are most efficient if they are based on detailed knowledge of the life history, biology, ecology and susceptibility of the pest species to eradication and control measures. Species-specific rapid response manuals have been prepared for several marine pests that the Marine Pest Sectoral Committee (MPSC) has identified as being of national concern.

During an emergency response, detailed technical information must be collected in the investigation phase of the response. At a minimum, information will be needed on:

- the nature of the pest, including its:
  - taxonomy
  - known distribution (global/Australian, native/non-native)
  - life history and ecology
  - environmental tolerances
  - impact potential
- pathways and vectors by which the species may be spread
- methods to prevent spread of the organism
- methods for undertaking surveys to
  - delimit established populations
  - trace an incursion
  - monitor the effectiveness of management measures
- methods to control or eradicate pest populations in different marine environments
- federal, state and territory legislation and policy relevant to emergency responses.

This information must be assembled rapidly from reliable sources. Preference should be given to using primary sources of information, such as advice from scientists, engineers or other professionals with recognised expertise on the species or likely emergency operations, and from published, peer-reviewed literature. Reputable secondary sources of information, such as internet databases and ‘grey’ literature may be used to supplement this advice or to prepare summary information and plans for expert review.

This document provides guidance on:

- types of information needed to determine an appropriate response to the suspicion or confirmation of incursion by *Undaria pinnatifida*
- types of expert advice that may need to be sought
- potential sources of information for preparing a response plan
- appropriate methods for containment, control and/or eradication of established populations.

1 Nature of the pest

Understanding the life history, ecology and biology (Table 2) of a marine pest is fundamental to an effective emergency response. Detailed knowledge of a species allows better evaluation of the threat it is likely to pose, the feasibility of response options and the design of efficient methods for surveillance, containment, eradication and control.

1.1 *Undaria pinnatifida*

The Japanese kelp or wakame, *Undaria pinnatifida* (Harvey) Suringar 1873, is an invasive species of brown macro algae that has been introduced to several continents worldwide. It occurs in lower intertidal and shallow subtidal habitats, often growing on non-natural structures such as wharf piles, mooring ropes and marina pontoons. In its native habitat, it occurs in dense stands, forming a thick canopy on a wide range of shores from low tide level to a depth of more than 15 m in clear waters (NIMPIS 2002). *U. pinnatifida* is listed on the Australian Priority Marine Pest List (APMPL) as a nationally significant marine pest species.

1.1.1 Taxonomy

Table 1 Taxonomy of *Undaria pinnatifida*

Classification	*Undaria pinnatifida*
Phylum	Heterokontophyta
Class	Phaeophyceae
Subclass	Laminariales
Order	Alariaceae
Family	*Undaria*

Undaria pinnatifida is a laminarian kelp. The order Laminariales is characterised by large size, a flattened lamina and a cylindrical compressed stipe. Adult plants (sporophytes) of *U. pinnatifida* superficially resemble the native laminarian kelp *Ecklonia radiata* (family Alariaceae), which occurs in shallow subtidal habitats of southern Australia (Womersley 1987). *U. pinnatifida* sporophytes can be distinguished from *E. radiata* by the presence of a central midrib along the lamina and the presence of a well-developed sporophyll in mature sporophytes, both of which *E. radiata* lacks (Photo 1).

Species that are similar to *U. pinnatifida* but do not occur in Australia include *Alaria esculenta* (Linnaeus) Greville 1830, *Saccorhiza polyschides* (Lightfoot) Batters 1902, *U. undarioides* (Yendo) Okamura 1915 and *Undariella peterseniana* (Kjellman) Y. Lee, 1998.

Alaria esculenta is a North Atlantic species that occurs on exposed rocky shores low in the intertidal zone. It has olive or yellow–brown fronds, which grow to 4 m long and 25 cm wide and which, like *U. pinnatifida*, have a distinct midrib. The lamina of *A. esculenta* is not split into pinnate lobes but can look lobed when in an eroded state.

Saccorhiza polyschides occurs naturally in the north-eastern Atlantic, from northern Africa to Scandinavia. The reproductive plants of *S. polyschides* have a fluted sporophyll just above the holdfast, like *U. pinnatifida*, but it can be distinguished from *U. pinnatifida* by the lack of a midrib. It has a deeply cleft lamina consisting of many linear segments. Both *U. undarioides* and *U. peterseniana* have similar native geographic ranges to *U. pinnatifida* (north-west Pacific) and similar gross sporophyte morphology.

1.1.2 Diagnostic features for identification

Undaria pinnatifida can be identified in the field and in the laboratory.

1.1.2.1 Field identification

Adult plants (sporophytes) of *Undaria pinnatifida* are usually 1.5 to 2 m in length but can reach up to 3 m. The sporophyte has a branched holdfast, a stipe (stem) and a flattened, lobed or divided blade with a distinctive central midrib. The stipe is very distinctive, having a corrugated appearance giving rise to a broad, flat lanceolate blade with a distinctive midrib (NIMPIS 2002). Reproductively mature sporophytes develop a characteristic fluted or corrugated structure (the sporophyll) on the stipe between the holdfast and blade (Photo 1). Juvenile *U. pinnatifida* sporophytes lack the sporophyll and can be difficult to distinguish from other species of brown alga until the midrib becomes visible on the blade (Photo 2). This generally occurs when the plant is over 5 cm in height.

Photo 1 Adult *Undaria pinnatifida* sporophyll

Image: CSIRO Marine and Atmospheric Research

Photo 2 Split frond of a young sporophyte showing pronounced midrib but no sporophyll

Image: K Seaward, Marine Ecology Research Group, University of Canterbury 2006

1.1.2.2 Laboratory identification

The sporophyte of *Undaria pinnatifida* can reach lengths of 1 to 3 m in most areas but it is usually less than 1 m in the Mediterranean Sea, along the Spanish coast and in some populations in New Zealand and Australia. Small sporophyte size is often associated with turbid waters. The stipe is attached by root-like haptera to the substrate, and the leathery-to-membranous lamina is yellow–brown to brown, becoming olive coloured when dry.

It is pinnate and young sporophytes lack a midrib (Photo 3). They can be distinguished from other kelps from about 1 cm in size because of their glandular cells, which are visible on close inspection as small dark dots. *U. pinnatifida* includes at least two morphological forms: *U. pinnatifida* f. *disticha* and *U. pinnatifida* f. *distans*. The latter has a long stipe and sporophylls that often do not reach up the lamina (Figure 1).

Figure 1 Diagnostic features of *Undaria pinnatifida*

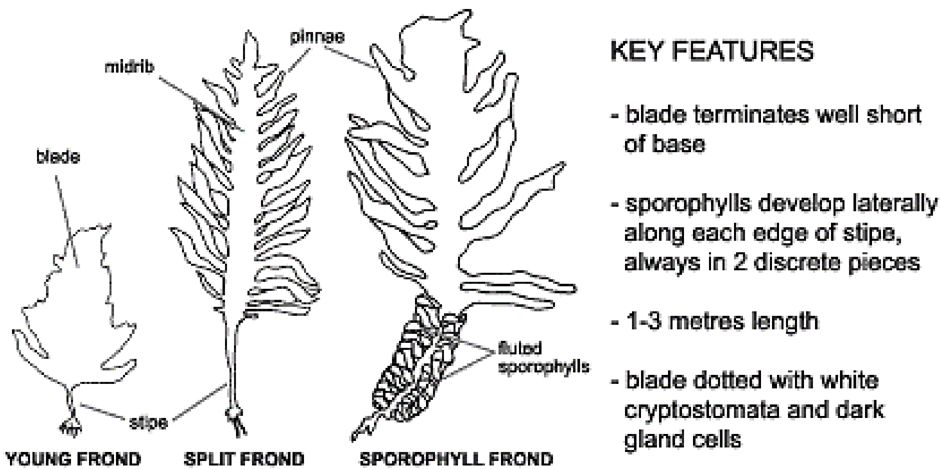


Image: Sanderson 1990

Photo 3 Pressed split frond with developed pinnae

Image: Aquatic Biodiversity & Biosecurity Update No. 20, November 2006

1.1.3 Life history and ecology

Understanding the ecology of *Undaria pinnatifida* involves examination of its reproduction, growth and life habit (Table 2).

Table 2 *Undaria pinnatifida* life history summary

Feature	Measure
Maximum size (length)	1–3 m
Maximum age (sporophyte)	1 year
Mating strategy	Sporic lifecycle
Type of mating	Alternation of heteromorphic generations
Dispersal stage	Motile zoospore/drifting sporophytes
Zoospore longevity	Up to 222 days
Gametophyte longevity	at least 2.5 years
Time to sexual maturity	50–70 days
Size at sexual maturity (length)	33 cm
Feeding mode	Photosynthesis
Depth range	Intertidal to approximately 20 m
Preferred habitat	Fouling vertical, hard substrata in sheltered environments
Distribution	Gregarious settlement
Salinity tolerance	Above approximately 27 ppt
Temperature tolerance (sporophyte)	3–23 °C

1.1.3.1 Reproduction and growth

Like all brown kelps, *Undaria pinnatifida* has a biphasic (sporic) lifecycle (Figure 2). It has separate haploid (gametophyte) and diploid (sporophyte) generations. The diploid (sporophyte) generation grows to become the visible plant. The haploid (gametophyte) generation is microscopic. The sporophyte stage of *U. pinnatifida* is mostly annual, germinating in late summer or autumn, growing rapidly throughout winter and spring and then senescing in mid-to-late summer (Schaffelke et al. 2005).

Mature sporophytes release motile zoospores from the fluted sporophylls located at the base of the stipe. Millions of zoospores may be released per gram of sporophyll tissue. The zoospores swim actively for 5 to 6 hours before settling (Hay & Luckens 1987), but they can remain viable for up to 222 days. Zoospores settle onto suitable substrata and develop into separate, microscopic male and female gametophytes. Male gametophytes release motile sperm, which fertilise eggs produced by the female gametophytes. The resulting zygote develops into the sporophyte (Thornber et al. 2004). *U. pinnatifida* is not known to reproduce vegetatively by fragmentation but it has been observed in the laboratory to reproduce asexually through unfertilised eggs, which have developed into parthenogenetic sporophytes.

Figure 2 Lifecycle of *Undaria pinnatifida*

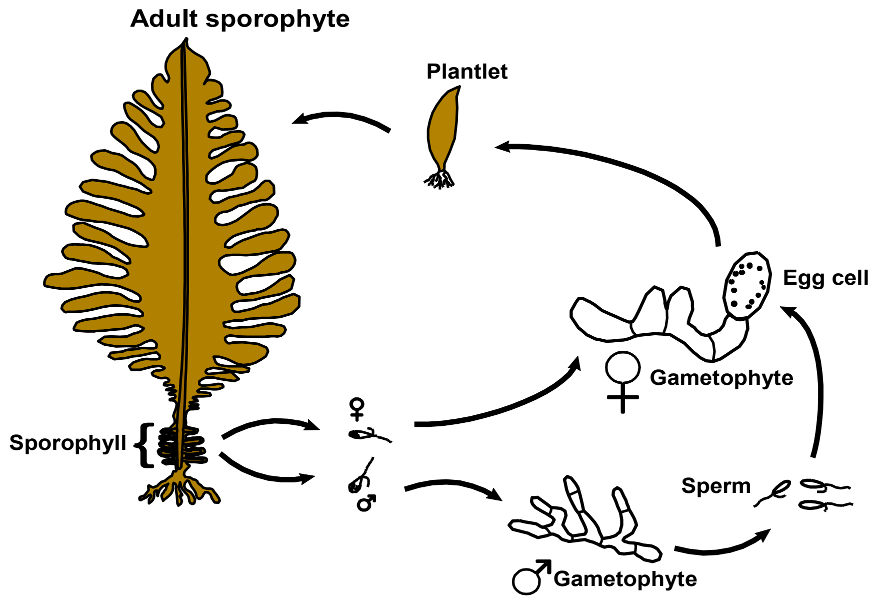


Image: Sinner et al. 2000a

In its native range in temperate Japan, China and Korea, small, microscopic sporophytes appear during winter, mature during the spring and senesce during the summer as water temperatures increase (Castric-Fey et al. 1993; Thornber et al. 2004). Fertilisation and early sporophyte development occur when water temperatures are between 10 and 20 °C (Akiyama 1965). Maximum maturation of female gametophytes (gametogenesis) of *U. pinnatifida* has been reported as occurring at 17 °Celsius, with a day length of 12 hours (Choi et al. 2005). *U. pinnatifida* gametophytes remain dormant during high summer water temperatures.

Other species of kelp in Australia have a gametophyte stage that persists over the winter months rather than remaining dormant over summer. In Tasmania, gametogenesis of *U. pinnatifida* may be possible throughout the year, with maximum rates around March and September (Hewitt et al. 2005).

At Tinderbox Marine Reserve in Tasmania, development into sporophytes of visible size was observed to take 20 to 30 days (Hewitt et al. 2005). Average growth in total length occurred at about 10 to 20 mm/day, but it may vary depending on the turbidity, height on shore, water depth at which the plant grows and availability of nutrients. Mature sporophytes with sporophylls were present at Tinderbox throughout the growth season, with peaks in abundance during summer (December to February).

U. pinnatifida sporophytes become mature (actively release zoospores) at a relatively small size. Plants as small as 33 cm in length can develop sporophylls. However, these very small mature sporophytes are typically present in very small numbers, and the number of zoospores they release is low in comparison to larger plants (Schaffelke et al. 2005). In Tasmania, zoospore release is limited to sporophytes of 55 cm or longer for most of the growing season, with the proportion of mature sporophytes increasing towards the end of the season. Small sporophytes with mature sporophylls were not observed until late in the growing season, after November.

In southern New Zealand, growth of sporophytes from recruitment to reproductive maturity takes around two to three months. Some populations in southern New Zealand exhibit two recruitment peaks per year, resulting in overlapping sporophyte generations (Thompson 2004). In these circumstances, some sporophytes can be found throughout the year, but in reduced abundance over the autumn months (Hay & Villouta 1993; Stuart 2004).

Microscopic stages of *U. pinnatifida* are present in the water column when the water temperature is warmest and pleasure boat traffic is greatest. Hewitt et al. (2005) also suggested that seed banks of microscopic stages (zoospores, gametophytes or sporelings) with significant longevity might be the source of continually recruiting sporophytes at Tinderbox Marine Reserve. It is not known how long the gametophyte stage can persist in natural conditions, but field studies have shown that sporophytes continue to recruit from microscopic stages for more than 2.5 years after the mature canopy of sporophytes has been removed (Hewitt et al. 2005).

Growth rates in the Sea of Japan (Russia) indicate that growth begins at temperatures as low as 0 °C, with maximal biomass increase during spring and a considerable slowing of growth at temperatures above 12 °C. The seasonal features of *U. pinnatifida* growth are the result of modulations in sporophyte morphology and sporulation, followed by disintegration of the blades (Skriptsova et al. 2004).

1.1.3.2 Life habit

Undaria pinnatifida is most commonly found on sheltered hard shorelines or substrata, from the lower intertidal depth to 15 m or more. The depth at which it occurs appears to be limited mainly by light and the availability of suitable substrata. The greatest densities of *U. pinnatifida* are usually found close to the surface, except on exposed coastlines and in clear waters. Off southern California (Santa Catalina Island), however, it occurs to a depth of 26 m, with highest abundances at 24 m.

Although *U. pinnatifida* has been described as colonising areas that are sheltered and rarely subject to significant wave action, it is capable of inhabiting a broader range of environments. Surveys of introduced populations in New Zealand have found stands of reproductive *U. pinnatifida* on wave-exposed shorelines amongst the large bull kelp species *Durvillaea antarctica* and *D. willana* (Russell et al. 2008). Its predominance in sheltered ports and harbours most likely reflects the distribution of founding populations.

Its large zoospore output, dormant microscopic stages and rapid growth make *U. pinnatifida* a highly opportunistic species. *U. pinnatifida* sporophytes grow readily on artificial substrata, including wood, bottles, ropes, tyres, boulders, cobbles, piles, loose gravel and various other hard surfaces (Hay & Luckens 1987).

U. pinnatifida is not highly competitive and does not usually displace native algal species through direct shading, but it does colonise areas that have recently been disturbed due to dieback, fishing pressure, grazing or species removal (Valentine & Johnson 2003; Valentine & Johnson 2004). Deterioration of a native algal canopy through storms or other disturbances can be rapidly followed by recruitment of large densities of *U. pinnatifida* sporophytes in disturbed patches (Valentine & Johnson 2004). The frequency and intensity of disturbance are therefore important in determining the resilience of native habitats to *U. pinnatifida* invasion.

Plant density can be variable depending on season and environmental factors. Maximum densities recorded in introduced populations range from 200 to 250 plants per m^2, and biomasses greater than 10 kg/m^2 (wet weight) have been found in New Zealand (Hay & Villouta 1993).

Temperature is the key influence on introduction and establishment of the species. In Asia, the lowest recorded temperature for zoospore release was 5 °C, the highest was 23 °C, and the range for gametophyte maturity and fertilisation was 5 to 28 °C (Floc'h et al. 1991). The gametophytes are relatively tolerant to temperatures up to about 30 °C (tom Dieck 1993).

Maximum maturation of gametophytes has been reported at a temperature of 17 °C and a day length of 12 hours. No gametogenesis was reported under continuous light (Pang 1996). Fertilisation and early sporophyte development occurs from 10 to 20 °C (Akiyama 1965; Bite 2001). Growth rates are generally enhanced at lower temperatures, with optimum temperatures as low as 5 to 10 °C but no growth below 3 °C (Hay & Villouta 1993). Thus, in Tasmania, for example, *U. pinnatifida* should be able to recruit throughout the year.

Lifecycle modelling, based on the temperature tolerances of the different life stages of *U. pinnatifida*, suggests that it may be able to survive throughout coastal areas of southern Australian (including Tasmania) and may be able to complete its lifecycle as far north on each coast as the Tropic of Capricorn (Hayes et al. 2007). This is because the dormant gametophytes have a relatively high maximum temperature tolerance and may be able to survive over the extended period of warm water temperatures.

U. pinnatifida generally prefers salinity greater than 27 ppt. This salinity is considered necessary for growth, but zoospores are able to attach to substrates at salinity above 19 ppt (Saito 1975). Light tolerance and depth preference vary with life stage and season. The sporophytes have a greater tolerance to low light levels than many other kelp species. *U. pinnatifida* gametophytes are able to survive in darkness for at least seven months, enabling long-distance transport in ballast tanks, and, if exposed to direct sunlight, the gametophytes will die within hours (Kim & Nam 1997). The light compensation point (the point above which net photosynthesis occurs) is low (8–15 microequivalents/m^2/s) and plants have very low respiration rates (Campbell et al. 1999). In some populations, gametophytes mature under both long and short day length, while others require short days.

Depending on the invasion location, *U. pinnatifida* is susceptible to a range of herbivores. In its native range in Japan, herbivorous fish exert significant grazing pressure on *U. pinnatifida* (Kiyomoto et al.

2000). In experiments examining teeth-shaped scars on the blades of *U. pinnatifida*, 93% of individuals had been grazed by *Calatonus japonicus* and 83% of individuals had less than half the normal blade area remaining (Kiyomoto et al. 2000). Sea urchins also graze on *U. pinnatifida*, and *Heliocidaris erythrogramma* has been recorded as destructively grazing sporophytes on urchin barrens. When sea urchins are removed from a heavily grazed area, *U. pinnatifida* slowly becomes the dominant canopy-forming species (Valentine & Johnson 2003).

1.1.4 Global and Australian distribution

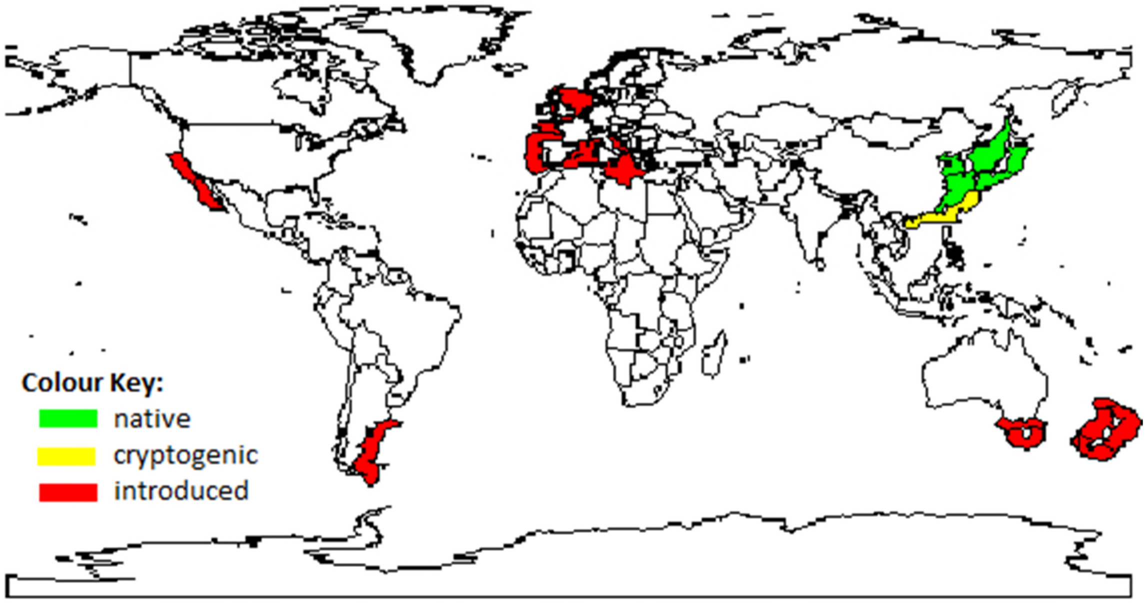
Undaria pinnatifida is native to Japan, Korea and China. In Australia, *U. pinnatifida* is known to be present in Victoria and Tasmania (Map 1). It was first recorded in Australia in 1988 at the port of Triabunna on the east coast of Tasmania (Hewitt et al. 2005; Sanderson 1990; Sanderson & Barrett 1989). It has since spread within Tasmania, occurring in high densities in the Mercury Passage and at the Tinderbox Marine Reserve (Sliwa et al. 2006). It is also present in Hobart (Hayes et al. 2007). Juvenile plants were found in Port Phillip Bay, Victoria, in 1996, on a rubble basalt reef, 3 m deep and covering an area of 1 to 2 km^2 (Campbell & Burridge 1998). It subsequently spread along the west coast of Port Phillip Bay between Point Wilson and Long Reef. Morphological and molecular differences indicate that the Victorian population is not derived from Tasmania, and is likely to have originated from northwest Pacific populations or as a secondary introduction from New Zealand (Campbell & Burridge 1998; Uwai et al. 2006).

A small population of *U. pinnatifida* was discovered on discarded shells at Flinders in Western Port, Victoria in 2000 (Parry & Cohen 2001). All sporophytes were removed from this location, and subsequent surveys in January 2001, May 2001 and February 2004 did not locate any further plants (Hayes et al. 2007). *U. pinnatifida* was also detected in Apollo Bay in Victoria (2009), and was subjected to an initial eradication attempt shortly after being found. However, when it was clear that eradication could not be quickly or easily achieved, management shifted to community engagement and control efforts to reduce further risk of spread.

In New Zealand, *U. pinnatifida* was first reported in Wellington harbour in 1987 (Hay & Luckens 1987). It then spread along the coastline to Oamaru and Timaru (1987 and 1988), Lyttelton (1991), other South Island east coast locations, the North Island of New Zealand and the Chatham and Stewart Islands (Hay & Luckens 1987). The most recent translocations include Tauranga Harbour and Port Taranaki (2005) (Russell et al. 2008).

The first record of *U. pinnatifida* along the European Atlantic coast was in the 1990s, but it was recorded in the Mediterranean Sea in 1971 (Curiel et al. 2001). In December 1992, *U. pinnatifida* was detected close to the international dock of Puerto Madryn, central Patagonia, Argentina. It was discovered in March 2000 on a floating pier and floating boom in Los Angeles Harbour, California, United States. It continued to spread and was found 500 kilometres north in Monterey Bay on more floating docks one year later (Silva et al. 2002).

Map 1 Global distribution of *Undaria pinnatifida*



Cryptogenic Unknown origin, may be native or introduced.
Source: NIMPIS 2002

1.1.5 Potential impact

The potential impacts of *Undaria pinnatifida* on native marine assemblages are of concern for many countries but are not yet well understood. Invasiveness is often related to a short lifecycle, uniparental reproduction, a broad ecological niche, genetic polymorphism, phenotypic plasticity and phylogenetic distance from native plants. *U. pinnatifida* exhibits most of these traits. The r-selected sporophyte stage is short-lived (six to nine months), and the species has rapid growth (1 cm/day) and matures early (at 50 to 70 days). It has a rapid uptake of nutrients, and a single plant is capable of releasing millions of spores during its reproductive period.

The establishment of strands of *U. pinnatifida* can lead to an increase in biodiversity in areas that were otherwise devoid of native assemblages, but in areas where diverse assemblages already occur it may displace native seaweeds and lead to a decrease in biodiversity and loss of spatial heterogeneity (Stuart 2004). It does not appear to be an aggressively competitive species, but it is highly opportunistic, taking advantage of disturbance events and naturally patchy habitats to establish dense stands and displace native species (Stuart 2004).

Sporophytes of *U. pinnatifida* can cause significant fouling on aquaculture facilities, increased weight on floating structures and increased labour costs due to the need for de-fouling of stock and equipment. The potential impact of *U. pinnatifida* on social, economic and environmental factors is outlined in Table 3.

After its introduction to New Zealand, *U. pinnatifida* was included as an unwanted organism in the New Zealand Biosecurity Act 1993. Many regional strategies have included attempts to eradicate or prevent the pest from spreading to other localities, especially to New Zealand marine reserves.

Table 3 Categories of potential impact caused by *Undaria pinnatifida*

Impact category	Description	Potential impact
Social amenity	Human health	No
Economy	Aquatic transport	No
	Water abstraction/nuisance fouling	Yes
	Loss of aquaculture/commercial/recreational harvest	Yes
	Loss of public/tourist amenity	No
	Damage to marine structures/archaeology	No
Environment	Detrimental habitat modification	No
	Alters trophic interactions and food webs	No
	Dominates/out-competes and limits resources of native species	Yes
	Predation of native species	No
	Introduces/facilitates new pathogens, parasites	No
	Alters bio-geochemical cycles	Yes
	Induces novel behavioural or eco-physical responses	No
	Genetic impacts—hybridisation and introgression	No
	Herbivory	No

Source: Hayes et al. 2005

2 Pest pathways and vectors

Undaria pinnatifida spreads by movement of the microscopic zoospore stage (gametophyte) or the mature plant (sporophyte). The motile zoospore stage can spread naturally, through movement of water currents away from an infested area, or by transport in seawater moved by humans from the infested site. Fertile sporophytes can also be dispersed naturally by water currents, either as drifting thalli or attached to unstable substrata such as cobbles or shells (Sliwa et al. 2006). Empirical observations and experiments suggest that spread through natural spore dispersal is likely to be metres to tens of metres per year, and drifting sporophytes may be dispersed over hundreds to thousands of metres per year, depending on the strength of water currents (Forrest et al. 2000; Russell et al. 2008; Sliwa et al. 2006).

Movement of fouled structures (including vessels, anchors, chain lockers, moorings, ropes, floats and aquaculture equipment) is the main pathway for introduction and spread of this species (Bax et al. 2002; NIMPIS 2002; Rajagopal et al. 2006; URS 2004) (Table 1). However, ballast water is also a potentially important vector for *U. pinnatifida* (Hay 1990; Lewis 1999). Viable *U. pinnatifida* zoospores can remain in the water column for up to 222 days and may be taken up in ballast water or internal spaces of vessels—for example, in bilge water or anchor wells. *U. pinnatifida* could, therefore, be spread domestically around Australia through ballast water exchange (URS 2004). However, ballast water regulation both internationally and domestically should reduce this risk significantly.

Similarly, the relatively hardy, dormant, microscopic gametophyte stage may be transported alive, either attached to hulls or in the ballast water tanks of large ships.

Table 4 Pathways and vectors for *Undaria pinnatifida*

Pathway	Description	Vector for spread
Biocontrol	Deliberate translocation as a biocontrol agent	No
	Accidental translocation with deliberate biocontrol release	No
Canals	Natural range expansion through man-made canals	No
Debris	Transport of species on marine debris (includes driftwood)	No
Fisheries	Deliberate translocation of fish or shellfish to establish or support fishery	No
	Accidental with deliberate translocation of fish or shellfish	Yes
	Accidental with fishery products, packing or substrate	No
	Accidental as bait	No
Individual release	Deliberate release by individuals	No
	Accidental release by individuals	No
Navigation buoys, marine floats	Accidental as attached or free-living fouling organisms	Yes
Plant introductions	Deliberate translocation of plants species (such as for erosion control)	No
	Accidental with deliberate plant translocations	No
Recreational equipment	Accidental with recreational equipment	Yes

Pathway	Description	Vector for spread
Scientific research	Deliberate release with research activities	No
	Accidental release with research activities	No
Seaplanes	Accidental as attached or free-living fouling organisms	Yes
Vessels	Accidental as attached or free-living fouling organisms	Yes
	Accidental with solid ballast (such as with rocks or sand)	No
	Accidental with ballast water, sea water systems, live wells or other deck basins	Yes
	Accidental associated with cargo	No

Source: Hayes et al. 2005

Fouling on vessels is the most likely source of introduction into Australian waters. Vessel biofouling includes all external wetted surfaces, including seachests, bilge keels, anode blocks, rudder pins, propellers, shaft protectors, echo sounder transducers and log probes. It also encompasses all internal surfaces and niches that are exposed to seawater, including anchor wells, chain lockers, bilge spaces, fishing gear, bait lockers, cooling water intakes, strainer boxes and internal pipe work (AMOG 2002).

Secondary incursions of *U. pinnatifida* are often associated with the presence of recreational boat ramps or moorings (Seacare 2003, cited in Hewitt et al. 2005). Movement of adult sporophytes occurs as biofouling on submerged, non-permanent structures. *U. pinnatifida* readily colonises hard artificial and natural substrata.

Because it can grow attached to shells, submerged ropes, buoys and other artificial substrata, *U. pinnatifida* can readily be transported with movement of aquaculture stock and equipment. This appears to have been a major pathway for the spread of *U. pinnatifida* around New Zealand and other countries to which it has been introduced (Forrest & Blakemore 2006). Translocation of *U. pinnatifida* by aquaculture operations may occur inadvertently through movement of the dormant, microscopic gametophyte or by transport of juvenile or mature sporophytes.

3 Policy and rationale for incursion response

The policy and rationale for an incursion response is based on the generic policy for incursion response to marine pests in Australian waters, the control or eradication strategy for *Undaria pinnatifida*, the policy on decision points and the policy on funding of operations and compensation. This chapter is an overview of marine pest emergency procedures and policy.

3.1 Generic policy for incursion response to marine pests in Australian waters

The National Environmental Biosecurity Response Agreement (NEBRA) establishes national arrangements for responses to nationally significant biosecurity incidents when there are predominantly public benefits. In the absence of a marine pest-specific deed, responses to marine pest incidents can fall under the NEBRA. The NEBRA provides a mechanism to share responsibilities and costs for a response when eradication is considered feasible and other criteria are met. The Biosecurity Incident Management System provides guidance on policies and procedures for the management of biosecurity incident responses, including responses to marine pest emergencies within Australian waters.

3.1.1 Commonwealth, state and territory authority responsibilities

Lead agencies in the response to a marine pest emergency must collaborate with the Consultative Committee on Introduced Marine Pest Emergencies (CCIMPE) in developing a National Biosecurity Incident Response Plan (NBIRP) as required under the NEBRA. CCIMPE will review the NBIRP and provide advice to the National Biosecurity Management Group (NMG), which will determine whether national cost-sharing arrangements should be activated. If the NBIRP and cost-sharing arrangements are approved, CCIMPE will help an affected jurisdiction implement an NBIRP. State coordination centres must be established with responsibility for strategically managing a marine pest incursion and for ensuring that community and/or industry involvement and communications are in place.

Depending on the circumstances, a local control centre with responsibility for managing field operations in a defined area may be established to enable an efficient and effective operational response. While close communication between a state coordination centre and a local control centre is imperative for effective conduct of any emergency response, it is important that strategic management (state coordination centre) and operational management (local control centre) roles be kept separate to optimise effective decision making and implementation during a national biosecurity incident response.

When a national coordination centre is established to help manage concurrent incursions in more than one jurisdiction, national coordination will be effected through consultation with CCIMPE representatives and relevant industry and community sector organisations, as appropriate.

3.1.1.1 Consultative Committee on Introduced Marine Pest Emergencies

CCIMPE provides national coordination for managing marine pest emergencies and comprises senior representatives from each Australian jurisdiction with coastal borders (the Australian Capital

Territory is not represented). CCIMPE is the national technical body that advises NMG whether an incursion by an introduced marine pest represents a marine pest emergency (in a national context), and coordinates the national technical response. CCIMPE also makes recommendations on possible stand-down phase activities (such as monitoring).

3.1.2 Emergency response stages

Management of a marine pest emergency of national significance has four phases of activation:

- investigation phase
- alert phase
- operations phase
- stand-down phase.

The first two phases, while detailed separately in the rapid response manuals, may be run concurrently, as outlined in the Biosecurity Incident Management System. Progression from one stage to the next depends on the nature of the emergency and available information.

Not all detections of marine pests will initiate a response involving all four phases and certain responses (such as detection of marine pests on vessels) may involve truncated responses.

3.1.2.1 Investigation phase

The investigation phase is in effect when relevant authorities are investigating a reported detection of a marine pest. The initial report of a suspected marine pest may come from port surveys, in water vessel inspections, slipway operators, fishermen, members of the public and routine field and surveillance activities.

A notifying party must advise CCIMPE of a suspected outbreak of a marine pest within 24 hours of becoming aware of it to be eligible for cost sharing under the NEBRA. When making a preliminary assessment, the notifying party may decide that a notification is likely to trigger a marine pest emergency alert when:

- the species detected is likely to be of national significance (Schedule 2 of the NEBRA) based on available data.
- the description matches a species represented on the APMPL that is either not present in Australia or, if it is present, the detection represents a new outbreak beyond the known range of established populations of the species in Australia. All APMPL species have been assessed to be of national significance.
- the species detected has a demonstrable:
  - invasive history
  - impact in native or invaded ranges on the economy, the environment, human health or amenity
- the suspected outbreak cannot be managed through pre-existing cost-sharing arrangements
- one or more relevant translocation vectors are still operating.

If the investigation indicates that a marine pest emergency is highly likely, the notifying party will inform the reporting point and will direct implementation of the alert phase.

Given that *U. pinnatifida* is already established in Australia and is on the APMPL, a suspected detection outside its current range will represent a possible range extension and trigger an emergency alert. If the subsequent investigation concludes that the situation does not constitute a marine pest emergency, the notifying party will inform CCIMPE and the emergency alert will be cancelled. However, ongoing actions to limit spread of the pest may be undertaken.

The CCIMPE Trigger List is currently under review but is still used for reporting purposes. It lists 35 marine pests of national concern for which a marine pest emergency response may be declared. CCIMPE may also consider an emergency response to marine pests not on the trigger list if they meet at least one of the National Environmental Biosecurity Response Agreement national significance criteria.

3.1.2.2 Alert phase

The alert phase is in effect while confirmation and identification of a suspected marine pest is pending, and an incident management team is assessing the nature and extent of the suspected incursion. During the alert phase:

- all relevant personnel are to be notified that an emergency alert exists in the affected jurisdiction
- an incident management team is appointed to confirm the identification of the suspected pest and to determine the likely extent of an incursion
- control measures are initiated to manage the risk of pest spread from affected sites (for example, operational boundaries of restricted areas are established for potential vectors)
- the findings of an emergency investigation are communicated to CCIMPE and NMG to enable a decision to be made on whether to proceed to the operations phase.

If an emergency investigation shows there is no incursion by a marine pest of concern or there is an incursion but it is unlikely to be eradicable, the notifying party will:

- ensure interim containment measures are implemented to minimise the risk of pest translocation from any infested waterway
- provide a situation report to the CCIMPE Secretariat for the information of CCIMPE representatives and request a CCIMPE teleconference to enable consultation with all jurisdictions
- on reaching agreement from CCIMPE, request that the transition to management phase (when there is a confirmed incursion by a marine pest of concern but eradication is not considered feasible)or stand-down phase be implemented (when investigation shows there is no incursion by a marine pest of concern)
- ensure documentation relevant to the decision-making process is maintained and filed as a ‘negative marine pest emergency alert’ (when investigation shows there is no incursion by a marine pest of concern) or a ‘non-eradicable marine pest emergency alert’ (when there is a confirmed incursion by a marine pest of concern but eradication is not considered feasible).

If the emergency investigation shows there is an incursion by a marine pest of concern and it is potentially eradicable, the notifying party will:

- ensure appropriate emergency containment measures are continued to minimise the potential for pest translocation, both from and within any infested waterway
- provide a situation report and an NBIRP plan to the CCIMPE Secretariat for urgent consideration by CCIMPE representatives and request a CCIMPE teleconference to enable consultation with all jurisdictions
- following CCIMPE endorsement, submit the NBIRP to NMG for consideration of national cost-sharing arrangements to help resource a national biosecurity incident response.

3.1.2.3 Operations phase

The Operations phase of an emergency response commences when the marine pest emergency is confirmed by agreement through the NMG forum and activities under a response plan are implemented. During the operations phase of a national biosecurity incident response:

- all relevant personnel and agencies should be notified that a national biosecurity incident response is being undertaken in the affected jurisdiction
- a standing committee on conservation and a local control centre should be established, if necessary
- control measures initiated in the alert phase should remain in place to manage the risk of pest spread from affected sites
- measures to eradicate the pest from infested sites should be implemented
- information from infested sites about the pest and the progress of operations should be collected, documented and analysed to enable progress of a national biosecurity incident response to be monitored
- expenditure associated with all eligible costs under cost-sharing arrangements should be documented
- regular situation reports should be communicated to the CCIMPE forum
- a decision should be made, when appropriate, on when to proceed to the stand-down phase.

3.1.2.4 Stand-down phase

The stand-down phase is in effect when, following appropriate consultation between the affected jurisdiction and CCIMPE, all agree that there is no need to progress or continue with a national biosecurity incident response. During the stand-down phase:

- a systematic approach to winding down operations must be taken to ensure operational effectiveness is not jeopardised
- all personnel, agencies and industry contacts involved in the emergency response are to be notified of the stand down.

The stand-down phase must commence once operational objectives have been achieved, or otherwise in accordance with advice provided by CCIMPE and agreed by NMG. The advice that an emergency eradication operational response is no longer needed must be communicated to the affected jurisdiction.

3.2 Control and eradication strategy for *U. pinnatifida*

Undaria pinnatifida is highly fecund and can form dense populations in intertidal and submerged marine habitats, where it can replace native Australian species. *U. pinnatifida* has the potential to cause nuisance fouling on marine infrastructure, with economic consequences for the aquaculture and maritime industries.

U. pinnatifida is present in Tasmania and Port Phillip Bay, Victoria; it is considered absent from all other Australian waters. Any reports of the suspected presence of *U. pinnatifida* in Australian waters should initiate the Investigation Phase of an emergency response.

The methods used to control an incursion of *U. pinnatifida* in Australian waters depend on the location and size of the outbreak. If the emergency investigation revealed an incursion by *U. pinnatifida* that was potentially eradicable, the Incident Manager would prepare an NBIRP and forward it to the CCIMPE Secretariat for urgent consideration.

The options for controlling an incursion by *U. pinnatifida* in Australian waters are:

1) Eradication of the pest from the infested area.
2) Containment, control and zoning with the aim of containing the species and slowing its further spread to other areas.

Eradication is unlikely if initial investigations show the species is widely established in open marine environments. Each control option involves a combination of strategies, such as:

- establishing declared areas to define zones where the pest is present or suspected to occur, and where emergency management operations are to be implemented
- quarantining and restricting or controlling movement of potential vectors, such as submersible equipment, vessels, marine organisms (fauna and flora) and ballast water in declared areas to prevent spread of the pest
- decontaminating potential vectors for the pest, including vessels, aquaculture stock and equipment, maritime equipment, and water that may contain larvae of the pest
- treating established populations on natural and artificial habitats in the infested area
- delimiting and tracing surveys to determine the source and extent of the incursion
- surveillance and monitoring to provide proof of freedom from the pest.

3.3 Policy on decision points

The policy on decision points includes proof of eradication and decisions to stand down eradication or control operations.

3.3.1 Proof of eradication

Proof of eradication requires a robust monitoring program during the operations phase of the response. During the operations phase, the purpose of the monitoring program is to detect new outbreaks of *Undaria pinnatifida* for treatment and to determine the efficacy of the treatment procedure. This information can be used to refine and direct treatment. Re-survey of treated sites is particularly important for *U. pinnatifida* incursions, as dormant microscopic gametophytes and embryonic sporophytes can remain viable and undetected for several years, allowing continuing recruitment and re-establishment of the population.

Monitoring should also continue at sites potentially at risk of infestation. A decreasing trend in the number of new, untreated populations of *U. pinnatifida* detected over time in the infested area is evidence of the effectiveness of control measures.

3.3.2 Stand down eradication or control operations

The optimal time to stand down monitoring, eradication and control operations is a trade-off between the costs of maintaining emergency operations, including ongoing surveys (Cs), the cost of escape (including likely impacts) if eradication is declared too soon (Ce), the probability of detecting the pest species given it is present (q) and the annual probability the species remains present (p). This rule of thumb can be used to calculate the optimal number of surveys:

$$n^* = \frac{\ln\left\{\dfrac{-C_s}{C_e \ln(r)}\right\}}{\ln(r)}$$

Where r = p(1 – q) is the probability the pest is not detected but is still present in the survey area. See Regan et al. (2006) for guidance on calculating this decision point.

3.4 Policy on funding of operations and compensation

CCIMPE will help determine whether an incursion is likely to be eradicable and when national cost-shared funding under the NEBRA should be sought. Cost-sharing must be agreed by NMG.

As detailed in the NEBRA, parties will share the eligible costs of emergency eradication responses as follows:

- a 50% share from the Australian Government
- a 50% share collectively from the states and the Northern Territory
  - this is calculated for each jurisdiction based on the length of coastline potentially affected by the species, and their respective human populations
  - only jurisdictions affected or potentially affected by the pest or disease are required to contribute.

NMG may commit up to $5 million (in annual aggregate) towards the eligible costs associated with an agreed national biosecurity incident response. If this $5 million is exceeded in any one financial year, NMG must seek ministerial approval from all parties to continue activities and/or begin new emergency responses.

Private beneficiary contributions to a response will be considered by NMG on a case-by-case basis where there is one or more private beneficiary and no existing arrangements.

4 Principles for containment, control and eradication

Eradication of incursions by *Undaria pinnatifida* depends on early detection and immediate action. Eradication is most likely to be successful in shallow waters or partially or fully enclosed waterways. In open coastal waters with moderate-to-high water exchange, individuals may be dispersed over a wide area. Where surveys indicate that an infestation is widespread, eradication action is unlikely to be successful.

Characteristics of this species and the pathways by which it is spread make it difficult to eradicate. These include:

- ability for mature plants (sporophytes) to produce both male and female zoospores, so a single plant is capable of producing viable offspring
- high fecundity, with single plants producing a motile zoospore stage that can be dispersed over large distances by water currents
- ability for mature plants to detach and drift with water currents over large distances, establishing new populations through release of zoospores
- ability to recruit and survive in both the macroscopic sporophyte and gametophyte life stages on vessel hulls
- ability for the microscopic gametophyte stage to persist in confined wetted spaces, including vessel ballast tanks, seachests, water intake pipes and bilge lockers, making detection difficult
- ability for both microscopic gametophytes and sporophytes to be transported with aquaculture stock or equipment.
- presence in estuarine environments, which can be turbid, making detection difficult.
- likelihood of being detected on non-commercial vessels from infested ports or marinas, whose movements are frequent and often difficult to trace.

The basis of eradication is rapid, effective quarantine of the infested area and any potentially contaminated vectors, and elimination of the pest where it is found.

4.1 Methods for preventing spread of the organism

Methods used to prevent the spread of the organism are quarantine and movement control, and treatment for decontamination of infested vectors.

4.1.1 Quarantine and movement controls

Quarantine and movement controls include an investigation phase, an alert phase and an operations phase.

4.1.1.1 Investigation phase

When the presence of *Undaria pinnatifida* is suspected in an area but a marine pest emergency has not yet been confirmed (see section 3.1.2.1), the notifying party should, when feasible, take steps to limit the spread of the suspected pest from the investigation site or area by initiating voluntary restrictions on movement of potential vectors. This may involve notifying relevant port authorities, marina operators, industry associations and vessel owners in the suspect site about the investigation into a possible marine pest emergency. Cooperation should be sought from these stakeholders to stop, restrict or inform the notifying party of movement of vectors from the site. Compliance with voluntary movement controls may be enhanced by distribution of appropriate public awareness materials about the pest.

The investigation phase should attempt to identify all potential vectors present at the site and their location. Possible vectors for the spread of *U. pinnatifida* are described in chapter 2.

4.1.1.2 Alert phase

If the initial investigation finds that *Undaria pinnatifida* is highly likely to be present (see section 3.1.2.2), the findings should be communicated to CCIMPE for consideration of the appropriate course of action to manage the risk of spread from affected sites. The incident management team must ensure appropriate measures are implemented. These could include:

- restrictions on movement of potential vectors, such as submersible equipment, fishing gear, vessels, marine organisms (fauna and flora) and ballast water into and out of suspect sites
- restrictions on benthic fishing, including bottom trawling, dredging, weighted line fishing and use of baited traps in potentially affected areas
- controlling movement of people (such as property owners, scientists, tourists) into or out of the suspect sites, as appropriate; this may include police involvement
- a hotline phone number for reported sightings of the pests and inquiries from affected parties
- tracing potential vectors that have left the site
- redirecting vessels that have already left the site to appropriate sites for inspection and/or decontamination, if appropriate
- requiring fishing vessels that have left the site to retain all seastar bycatch and shell debris until it can be inspected and cleared
- notifying and, where appropriate, consulting relevant experts.

4.1.1.3 Operations phase

The operations phase will be guided by whether eradication of the marine pest of national concern is feasible or not feasible.

Eradication not feasible

If investigation reveals an incursion by *Undaria pinnatifida* that is unlikely to be eradicable, interim containment measures (to prevent translocation of a pest of concern from any infested waterway) should be implemented to minimise the risk of the pest being spread from the infested area. A stand-down phase may be entered either directly from the alert phase or from the operations phase when CCIMPE and NMG agree there is no need to initiate a national biosecurity incident response.

Eradication feasible

If investigation reveals a potentially eradicable *U. pinnatifida* incursion, quarantine and associated movement restrictions can be implemented.

Quarantine restrictions require establishing specified areas:

- infested area—all or part of a waterway in which a marine pest emergency is known or deemed to exist (pending confirmation of pest identification)
- dangerous contact area—an area close to an infested area in which a pest has not been detected but, due to its potential for infestation, will be subject to the same movement restrictions as an infested area
- suspect area—an area relatively close to an infested area that will be subject to the same movement restrictions as an infested area (pending further investigation)
- restricted area—a defined area around an infested area that is subject to intensive surveillance and movement controls on potential vectors[2]
- control area—a defined area surrounding a restricted area in which biosecurity conditions apply to the entry or exit of potential vectors or specified risk items[2].

Similar terminology is applied to potentially affected vectors within each area. For example, a vessel within a dangerous contact area would be classified as a 'dangerous contact vessel'; a vessel within an infested area would be classified as an 'infested vessel'.

[2] Note that the legislative ability and scope of powers to establish biosecurity restricted areas and control areas will depend on the biosecurity legislation that is applicable within the relevant jurisdiction.

The extent of each specified area for *U. pinnatifida* should be determined based on:

- an initial delimiting survey of the area (section 5.3)
- an evaluation of the length of time the species has been present and whether it has reproduced; this would be based on the size and distribution of the animals in the infested area, the number of cohorts apparent and, when possible, examination of reproductive tissue
- the strength and distribution of directional or tidal currents
- expert advice.

Movement restrictions include limiting:

- the movement of vessels, immersed equipment, aquaculture stock or equipment and other vectors for biofouling
- fishing activities within the control area
- the uptake or movement of ballast water or other water from within the control area where appropriate controls are not in place.

Implementation of restrictions will be a dynamic process, determined by the location and extent of infestation and whether the aim is to eradicate the pest or to control its spread. Some restrictions may be deemed impractical or unnecessary in a particular circumstance, but others will be critically important to eradication or control.

Restricted Area Movement and Security Unit

The Restricted Area Movement and Security Unit of the Operational Pest Control Centre is responsible for controlling movement of goods, submersible equipment, vessels, water and other vectors including people into, within and out of the restricted area as appropriate to minimise the potential for pest spread. The unit's main duties are to:

- issue movement permits to the public
- establish and operate road and water checkpoints in the restricted area, including liaison with state transport authorities, water authorities, police and local government
- coordinate movement and security activities across infested sites
- maintain registers of all movements (in restricted and infested areas), permits issued and staff deployed.

Experience of movement controls

The emergency response to the incursion by the black striped mussel, *Mytilopsis sallei*, in Cullen Bay Marina (Darwin) in 1999, used a combination of the powers in the *Fisheries Act 1988* (NT) and the *Quarantine Act 1908 (Cwlth)* (superseded by the *Biosecurity Act 2015)* to impose sufficient quarantine measures to limit the spread of the species. The *Biosecurity Act 2015* can be used in the absence of appropriate state or territory legislative powers and may be used in circumstances, including directing conveyances[3]:

- into port
- to not enter a port and to obey further instruction
- to undergo a treatment action the Incident Manager deemed necessary.

The Australian Director of Biosecurity (or their delegate) can authorise State and Territory officers as biosecurity officers under the *Biosecurity Act* which will enable certain actions to be undertaken in a biosecurity response. All actions taken against a conveyance should only be taken in relation to those identified as being at risk of spreading the invasive species (Ferguson 2000). Guidelines for using the *Biosecurity Act 2015* are in Appendix A. The Biosecurity Act is only intended to be used if there is no appropriate State and Territory legislation that provides appropriate powers necessary for the response, aside from ballast water which is entirely covered by the Biosecurity Act. A provisional list of other Commonwealth and state powers for intervention and detention of vessels is in Appendix B.

Each state and territory should consider enacting relevant fisheries or other legislation to prevent or control fishing within a control area, and prevent or control translocation of stock and equipment from within it. Any requested movement of fishing gear or aquaculture stock or equipment should be subject to risk assessment consistent with procedures in the National Policy Guidelines for the Translocation of Live Aquatic Organisms (Department of Agriculture 2020). All potentially infested fishing gear, aquaculture equipment or stock should be treated and inspected before removal from the control area.

4.1.2 Surveillance for high-risk vectors

In the event of an emergency marine pest response, movement controls on potential vectors and pathways will be easier to manage if efforts can be targeted at vectors that pose the greatest risk of spread.

All vessels and other vectors that have been within an infested or dangerous contact area during the time the pest is known or suspected to have been present should be considered at high risk of transporting the pest. Vessels, oil rigs, barges and other moveable structures that have been present in suspect, restricted or control areas, should also be treated as high risk. The risk status of vessels may be changed if inspections or surveys find no sign of the pests.

Vessels that have not been within the infested or dangerous contact areas, but which have been in close proximity to a high-risk vessel that have departed these areas or the control area should also be considered high risk. All high-risk vessels should be required to proceed to an approved inspection and treatment facility.

[3] Under the *Biosecurity Act* the definition of conveyances includes vessels and floating structures

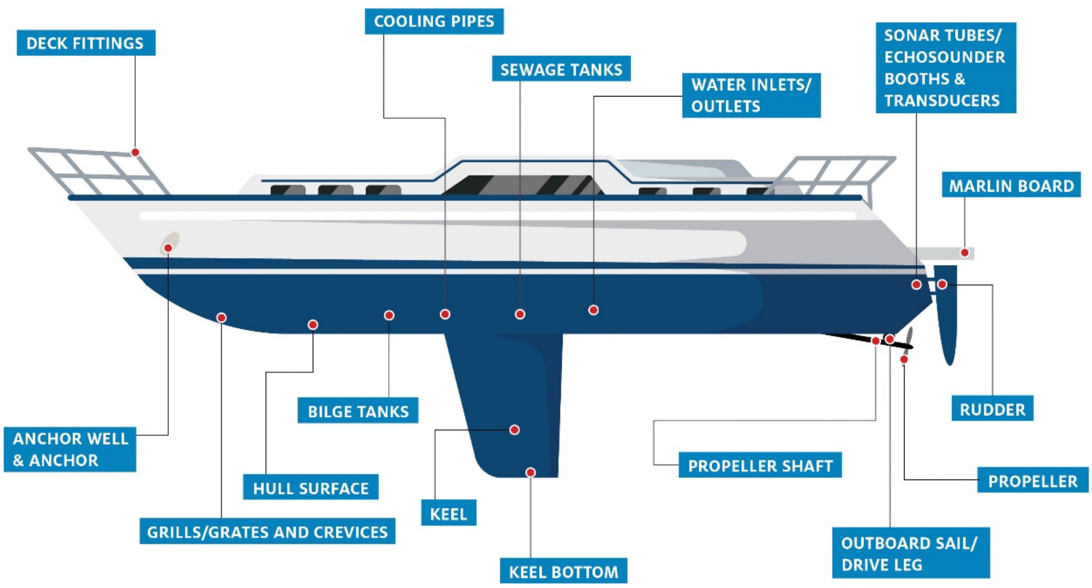
Where resources allow, all vessels and potential vectors within the control area should be inspected for signs of the pest. Medium-risk vessels should be required to remain within the control area until they can be inspected and declared free of the pest.

Divers or Remotely Operated underwater Vehicles (ROVs) should perform in-water inspections using a standardised search protocol. Biofouling is likely to be greatest in wetted areas of the vessel that are protected from drag when the vessel is underway and/or where the antifouling paint is worn or damaged or was not applied.

For vessels smaller than 25 m in length (Figure 3), attention should be focused on inspecting:

- rudder, rudder stock and post
- propellers, shaft, bosses and skeg
- seawater inlets and outlets
- stern frame, stern seal and rope guard
- sacrificial anode and earthing plate
- rope storage areas and anchor chain lockers
- ropes, chains or fenders that had been left over in the water
- keel and keel bottom
- sounder and speed log fairings.

Figure 3 High-risk niche areas for inspection of biofouling on vessels less than 25 metres



For vessels larger than 25 m in length (Figure 4), additional high-risk niche areas include:

- dry docking support strips (DDSS)
- seachests and gratings
- sonar tubes
- bow thrusters
- keel and bilge keels
- ballast tanks and internal systems.

Figure 4 High-risk niche areas for inspection of biofouling on vessels greater than 25 metres

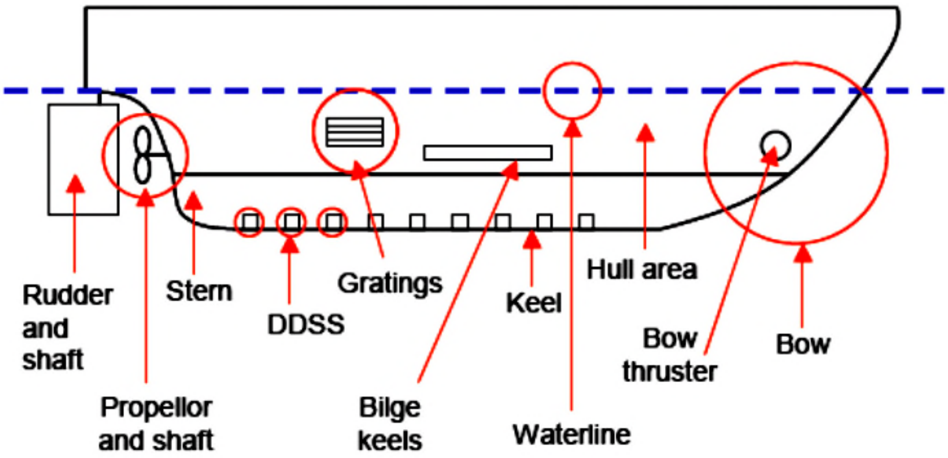


Image: Floerl 2004

Divers can inspect interior spaces and crevices (such as seachest, water intakes or outlets) using endoscopes.

All high-risk and medium-risk vessels that have recently left a control area should be contacted immediately. If they have not entered another port or marina they should be encouraged to remain at sea, no closer than 1.5 nautical miles to the nearest land until inspection and/or quarantine arrangements can be made. Biosecurity risks detected before or during this inspection must be dealt with before the vessel can be brought further inshore. Where the vessel has entered another port or coastal area, it should be inspected immediately and, if signs of the pest are present, the vessel should be directed for treatment, a back tracing of the vessel's itinerary be done and surveys undertaken of the anchorages it has visited.

Vessels that have been inspected and found to be free of the macroscopic sporophytes of *U. pinnatifida* should continue to be monitored over the potential growing season of *U. pinnatifida* for signs of germination of sporophytes from microscopic life stages.

4.1.3 Treatment methods for decontaminating infested vectors

Treatment methods differ depending on the type of area in which the infestation occurred. It could have been found in ballast water, on vessels or on equipment and marine organisms.

Table 5 summarises management recommendations for different types of vectors.

Table 5 Management recommendations for different types of vectors

Potential vector	Suggested management
International and domestic yachts and other vessels smaller than 25 m	Clean external submerged surfaces Treat internal seawater systems Manage ballast water Remove from the control area once cleaned
Domestic fishing vessels, ferries, tugs, naval vessels	Clean external submerged surfaces Treat internal seawater systems Manage ballast water
Merchant vessels larger than 25 m departing for other Australian destinations	Inspect and (where possible) clean external submerged surfaces Treat or seal internal seawater systems Manage ballast water
Merchant vessels larger than 25 m departing for international waters	Inspect and (where possible) clean external submerged surfaces Treat or seal internal seawater systems Restrict uptake of ballast water from the control area Manage ballast water
Recreational craft (such as dinghies, jet-skis, kayaks, outboard motors)	Clean external submerged surfaces Clean and dry internal seawater systems Educate users and service agents of risk
Fishing gear and nets	Clean and dry on removal from area Educate users of risk
Aquaculture stock (shellfish)	Remove from infested area, declump and immerse in 2% detergent (DECON 90) for 30 minutes; rinse in seawater and hold in quarantine facilities before re-deployment into marine environments
Aquaculture equipment	Remove from infested area Clean thoroughly by high pressure (greater than 2,000 psi) water blasting Immerse in 2% detergent solution for eight hours, or 3% liquid sodium hypochlorite for two hours, or hot water (40 °C) for at least one hour Rinse in sterile seawater and air dry
Buoys, pots, floats	Clean and dry Restrict removal from the control area Educate users on risks
Water, shells, substratum, live hard-shelled organisms from the control area (such as aquaria, bait)	Restrict removal from the control area Educate users on risks
Flotsam and jetsam	Remove from water/shoreline Dry prior to onshore disposal If possible, use barriers to prevent escape from infested area
Fauna (such as birds)	Verify the importance of the vector during delimitation surveys

Potential vector	Suggested management
Stormwater pipes, intakes	Clean Where possible, seal until stand down of emergency response

Source: Bax et al. 2002

4.1.3.1 Ballast water

In the event of an emergency response, all ballast water sourced from the area would be considered high-risk to the Australian marine environment. The *Biosecurity Act*, which implements the International Convention for the Control and Management of Ship's Ballast Water and Sediments (Ballast Water Convention) together with the *Biosecurity (Ballast Water and Sediments) Determination 2017* (Ballast Water Determination), prohibits discharge of ballast water anywhere within Australian seas[4], subject to certain exceptions.

All vessels that contain ballast water will need to be appropriately managed according to the Australian Ballast Water Management Requirements. This includes via an approved method of ballast water management, or disposed of safely, such as through an approved ballast water reception facility. If *Undaria pinnatifida* is present in an area, steps can be taken by the Department of Agriculture to ensure no low-risk exemptions to discharge ballast water would be granted under section 23 of the Ballast Water Determination.

Since the Ballast Water Convention has come into effect, certain ships are no longer allowed to manage ballast water through exchange. These vessels are required to install acceptable ballast water management systems to ensure appropriate treatment of ballast water on-board. These systems eliminate harmful pests from ballast water by using methods such as UV treatment or chlorination. Vessels that are allowed under legislation to meet ballast water management requirements through exchange (subject to certain exemptions), would be required to conduct ballast water exchange outside Australia's 12 nautical mile territorial sea limit. Additional measures may need to be investigated where vessels utilise ballast water exchange and operate exclusively within a declared Same Risk Area, detailed within the *Biosecurity (Ballast Water Same Risk Area) Instrument 2017*.

[4] Under the Biosecurity Act, the definition of Australian seas changes depends on the Administration (the country's flag under which the vessel is registered) of the vessel. For Australian or foreign vessels whose Administration is party to the Ballast Water Convention, Australian seas is waters within the outer limits of Australia's exclusive economic zone (EEZ) (200 nautical miles from the territorial sea baseline). For other vessels, Australian seas is the waters within the outer limits of the territorial seas of Australian (12 nautical miles from the territorial sea baseline).

Operators may choose to retain high-risk water within a ballast water tank if there is no intention to discharge the water in Australian seas. However, carrying high-risk ballast water into Australian seas is strongly discouraged, as a vessel's itinerary may change, or discharge may be necessary in the case of safety or pollution considerations.

4.1.3.2 Biofouling of vessels and other possible vectors

Mechanical removal of biofouling on vessels includes land-based treatment, internal seawater systems and various in-water treatments.

Land-based treatment

Because *Undaria pinnatifida* is capable of inhabiting internal piping and water intakes that are not readily inspected underwater, haul-out of vessels and other non-permanent structures (such as moorings, pontoons and ropes) for inspection and treatment on land is the preferred option for decontamination. This may only be possible for vessels smaller than 25 m in length where suitable haul-out or dry-dock facilities are available within or in close proximity to the control area. Larger vessels may need to be inspected and treated in the water.

Internal seawater systems

There is a risk that sporophytes dislodged or disturbed during haul-out or cleaning of a vessel may release zoospores or remain viable and start a new population if returned to the sea. The biosecurity officer must approve haul out facilities used for decontamination. Such facilities should be fully contained so material removed from vessel hulls cannot return to the marine environment by any means, including direct disposal, run-off and aerosol drift. All macro (greater than 1 mm) particles removed from vessels cleaned out-of-water should be retained and disposed of in landfill (or as biohazard material if appropriate). All liquid effluent (runoff) from out-of-water vessel water-blasting or cleaning should be collected for treatment in a liquid effluent treatment system.

Woods et al. (2007) provide guidance for identifying vessel cleaning facilities suitable for removal of marine pests. Approved facilities should also comply with relevant state requirements for waste containment and disposal from slipways, boat repair and maintenance facilities.

High-pressure water blasting followed by prolonged (more than 7 days) aerial exposure may also be used to treat other fouled structures removed from an infested area (such as mooring blocks, pontoons, floats and fenders). However, materials such as ropes that have fine interstices, which may be protected from the blasting and which can retain moisture, should be treated chemically or be disposed of to landfill.

Internal seawater systems should be cleaned to the greatest extent possible. Treatments that have been shown to be effective in the laboratory for treatment of *U. pinnatifida* spores, germling sporophytes and gametophytes include immersion in:

- 2% bleach solution (active ingredient, 3% sodium hypochlorite) in water (preferably fresh) for one hour or longer (Gunthorpe et al. 2001)
- 2% detergent solution (DECON 90, active ingredient, less than 3% potassium hydroxide and anionic and non-ionic surfactants) in water (preferably fresh) for four hours or longer (Gunthorpe et al. 2001)

- fresh water at 20 °C for at least 48 hours (Forrest & Blakemore 2006)
- water at 35 °C for at least 45 minutes (Forrest & Blakemore 2006).

Other treatments have been shown to be effective for seawater systems for mussels (*Mytilus galloprovincialis planulatus*), which are considerably more resistant to environmental stresses than *U. pinnatifida* and are likely to also be effective. These include:

- 5% (by volume) industrial detergent Conquest or Quatsan in water (preferably fresh) for 14 hours (Lewis & Dimas 2007)
- chlorine concentrations of 24 mg/L for 90 hours (Bax et al. 2002)
- 1 mg/L copper sulphate solution for 38 hours (Bax et al. 2002).

The Incident Manager may approve other treatments.

The Australian Pesticides and Veterinary Medicines Authority may need to approve use of bleach, detergent, copper sulphate or other toxic chemicals for biofouling control. These can cause handling and disposal difficulties if used in large quantities (Lewis & Dimas 2007).

In-water cleaning

The Anti-fouling and in-water cleaning guidelines (2015) state that where practical, vessels and moveable structures should be removed from the water for cleaning, in preference to in-water operations. When removal is not economically or practically viable, the guidelines accept in-water cleaning as a management option for removing biofouling, provided risks are appropriately managed.

Applicants who wish to perform in-water cleaning in Australian waters should familiarise themselves with the principles and recommendations contained in the guidelines. In Commonwealth waters, applicants should first check their obligations under the Environment Protection and Biodiversity Conservation Act 1999 (EPBC Act). If the activity does not need to be referred under the EPBC Act, then applicants should self-assess their activity using the decision support tool in Appendix A of the Anti-fouling and in-water cleaning guidelines (2015). Applicants who wish to perform in-water cleaning in state or territory waters should contact the relevant agency in each state or territory jurisdiction for advice.

Vacuum and brush cleaning

The most commonly available in-water cleaning technologies are brushing and scraping, use of soft cleaning tools, and water or air jet systems. These methods vary in their effectiveness for removing and containing biofouling organisms, and in their suitability for use on different anti-fouling coating types. Further information about these cleaning methods can be found in the Anti-fouling and in-water cleaning guidelines (2015).

Rotating brush and vacuum systems for removal of fouling pests have been trialled in New Zealand (Coutts 2002). Preliminary results suggest that these systems remove a large proportion (greater than 90%) of low-to-moderate levels of fouling and collect, on average, over 90% of the material that is removed. Problems associated with vacuuming include the potential dislodgement of fouling organisms by dragging hoses and divers, reduced efficiency with variable hull shape and blockages by large organisms when there is heavy fouling. These dislodgements and blockages can cause damage to filter valves. Because of this, large sporophytes with sporophylls should carefully be removed manually before using brush and vacuum systems. Gametes of *U. pinnatifida* are microscopic and therefore filtering systems must be able to deal with their removal (Coutts 2002). Because of these problems, brush and vacuum systems should be used only where there are no other options for vessel treatment.

Wrapping and encapsulation

Methods for treating biofouling include wrapping and encapsulation and chemical treatment. Unlike vacuum and brush cleaning, these methods do not remove fouling from the submerged surface of the vessel and moveable structure but aim to kill the biofouling organisms.

Wrapping and encapsulation of the submerged surfaces of vessels using impermeable barriers, such as polyethylene plastic, have been used to treat fouling on vessels of up to 113 m long (Mitchell 2007). The wrapping deprives fouling species of light and food while continued respiration and decomposition of organisms within the barrier depletes dissolved oxygen in the water, thus creating an anoxic environment that is eventually lethal to all enclosed organisms.

Polyethylene silage plastic wrap (15 by 300 m, 125 µm thick) is cut to size to suit the vessel type and is deployed by divers in association with a topside support team. The plastic is passed from one side of the vessel to the other, overlapped and secured tightly using PVC tape or ropes to create a dark, anaerobic, watertight environment. Sharp objects on the hull (such as propeller blades) should be wrapped separately or covered with tubing or cloth before encapsulation to prevent tears in the plastic.

Properly deployed, the wrap should contain the pest species and its larvae; care should be taken to ensure that biofouling is not dislodged when the wrap is deployed. The wrap must remain in place for at least seven days to ensure mortality. Wrapping of vessels larger than 25 m in length is labour intensive and may take up to two days to deploy per vessel. In addition, the time needed for effective treatment (seven days) may be too slow when rapid treatment and turnaround of vessels is crucial.

This method of treatment is only suitable in relatively sheltered environments with slow current flow, since strong currents create difficulties in deploying the wrap and increase the chances of tears in the plastic.

Where very large vessels or several vessels need to be treated, the encapsulation technique will generate large amounts of plastic waste. Wrap and equipment used to deploy it must be disposed of in landfill or an approved solid waste treatment facility.

Commercial encapsulation tools are available, which can be applied to a vessel arriving in port, or to a vessel at anchor, alongside a wharf or in a marina berth.

Relevant agencies in each state or territory jurisdiction should be consulted about the suitability of a wrapping and encapsulation method for a vessel or moveable structure.

Chemical treatment

Mortality can be accelerated by adding chemical agents to the encapsulated water (Coutts & Forrest 2005). For example, sodium hypochlorite (NaOCl, 12.5% w/v) can be added to the sea water enclosed in the sheath to achieve a concentration of 200 to 400 ppm. The sheath and chemical treatment remain in place for 36 to 48 hours for each vessel. Because this technique may release some chloride ions to the surrounding water, consent is required from relevant state or territory authorities to undertake the treatment.

4.1.3.3 Aquaculture stock and equipment

Several treatments have been evaluated to remove *Undaria pinnatifida* from aquaculture operations. The biphasic lifecycle of *U. pinnatifida* means that treatments must effectively remove both the macroscopic sporophyte and any microscopic stages (zoospores, gametophytes or germling sporophytes) that may be present on the surface. Large, visible sporophytes can be removed manually, but infested surfaces must be treated to remove microscopic stages. Like other species of algae, *U. pinnatifida* can be susceptible to a wide range of treatments. However, treatment suitability and cost-effectiveness must be considered when attempting eradication or even to limit the spread of *U. pinnatifida* from established populations.

Treatments that have been trialled to remove *U. pinnatifida* from ropes, culture lines and equipment include:

- immersion in or spraying with:
  - acetic acid—4% (Forrest & Blakemore 2006)
  - chlorine or sodium hypochlorite (Gunthorpe et al. 2001)
  - detergent—2% (potassium hydroxide) (Gunthorpe et al. 2001)
  - brominated micro-biocide—Amersperse 261-T, Ashland Chemical (Stuart 2004)
  - hot (50 °C) or cold (ambient) freshwater (Forrest & Blakemore 2006; Gunthorpe et al. 2001; Webb & Allen 2001)
- air drying (Forrest & Blakemore 2006; Gunthorpe et al. 2001)
- high pressure (greater than 2,000 psi) water blasting (Forrest & Blakemore 2006).

Table 6 is a summary of treatments shown to cause 100% mortality (LD100) of *U. pinnatifida.* These are largely based on laboratory trials on microscopic life stages and must be adapted to ensure complete mortality on more complex structures such as ropes or nets.

Table 6 Treatments that achieved total mortality (LD100) of *Undaria pinnatifida* in laboratory conditions

Treatment	Duration of immersion and concentration for 100% mortality
Freshwater immersion	8 hours at 18 °C[a]
	10 minutes at 35 °C[b]
	45 seconds at 45 °C[b]
	5 seconds at 55 °C[b]
Acetic acid	1 minutes at 4% in fresh water[b]
Air-drying	3 days at 10 °C (55–85% humidity)[b]
	1 day at 20 °C (55–85% humidity)[b]
	8 weeks at 10 °C (greater than 95% humidity)[b]
	6 weeks at 20 °C (greater than 95% humidity)[b]
Bleach solution (Black and Gold)[c]	1 hour at 2% concentration[a]
Detergent (DECON 90)[d]	>30 mins @ 2% concentration >18 °Ca

a Gunthorpe et al. 2001. Forrest & Blakemore 2006. **b** Forrest et al. 2007. **c** active ingredient 3% sodium hypochlorite. **d** active ingredient less than 3% potassium hydroxide.

Ropes and equipment

The protocols recommended for treating ropes and aquaculture equipment, such as buoys, floats, nets and traps, are:

1) Remove to land, taking care not to dislodge or disturb plants, to prevent release of zoospores, when removing the structures from the water.
2) Clean thoroughly by high pressure (greater than 2,000 psi) water blasting.
3) Immerse in 3% liquid sodium hypochlorite for at least two hours.
4) Rinse in seawater and air dry.

Aquaculture stock

Some cultured species with hard shells (such as molluscs and crustaceans) may be fouled by *U. pinnatifida* and, therefore, be potential vectors for their spread. Utility of methods used to decontaminate aquaculture stock will depend on the relative robustness of cultured stock to the treatment. For example, adults of thick shelled bivalves, such as oysters, are more resistant to treatment by freshwater immersion, chemicals, hot water and high-pressure water than *U. pinnatifida* (Forrest & Blakemore 2006; Forrest et al. 2007; Gunthorpe et al. 2001). Spat and less calcified juvenile oysters will not be as resistant to these treatments (Rajagopal et al. 2003).

A combination of the treatments described in Table 5, applied either at the same time or sequentially, may be most effective at removing all life stages of *U. pinnatifida* but may also cause greater stress to the treated stock.

Treatments recommended for removing *U. pinnatifida* life stages from bivalve stock, based on the results of Gunthorpe et al. (2001), Forrest and Blakemore (2006) and Forrest et al. (2007), include:

- declump stock, then immerse in fresh water at 18 °C or higher for at least 24 hours and air dry overnight
- declump stock, then immerse in 4% acetic acid for at least one minute, then rinse in seawater and air dry overnight
- declump stock, then immerse in 2% detergent (DECON 90) for at least 30 minutes, then rinse in seawater and air dry overnight.

All three protocols resulted in 100% mortality of *U. pinnatifida* but caused only minor changes in survival and viability of mussel stock.

These methods are also likely to be cost-effective in treating other fishing, aquaculture or boating equipment for *U. pinnatifida*.

Disinfection of bivalves and other aquaculture stock for external hitchhikers is not always effective and must be weighed against the potential environmental impacts of any treatment and effect on the stock. When a treatment cannot be effective, it may be precautionary to either destroy any potentially contaminated stock and dispose of it to landfill or harvest and process stock for human consumption

4.2 Tracing an incursion

Tracing is used to discover the method and pattern of the spread of the pests and may include trace-forward and trace-back. It is crucial to defining and modifying the dimensions of the specified areas and requires investigations that determine:

- the length of time the species has been present
- the initial source and location of infestation
- whether the pest has reproduced
- the possible movement of water, vessels, animals, submersible equipment and other potential vectors for the pest
- the existence and location of other potentially infested areas.

If the Local Control Centre is established, it is responsible for managing tracing and surveillance activities within the control area.

The cryptic nature of the gametophytes and germling sporophytes can make it difficult to establish the true state of a population of *Undaria pinnatifida*, including how widely distributed it may be. Because of the difficulties in sampling these microscopic stages in the wild, the population size and distribution is usually inferred from the distribution of visible sporophytes.

Several methods are useful for estimating how long the plants may have been present. Elements of the demography of the population may be inferred from the size distribution and reproductive state of individuals collected during the initial investigations. For example, *U. pinnatifida* that have produced a sporophyll may have been present for at least two to three months. The reproductive state of the sporophyll can be determined by excising a small portion of tissue and drying it for two hours in a cool, dark place in the laboratory. Spore release is then induced by rehydrating the tissue and agitating it in filtered seawater. Sub-samples of the seawater are then pipetted onto a slide and examined under a microscope for the presence of spores (Thompson 2004).

A population that contains sporophytes that vary widely in size, from mature plants with sporophylls to new germlings, may indicate successful local reproduction and multiple recruitment events.

4.2.1 Data sources for tracing vectors

Vessels

Tracing the movements of vessels to and from an incursion is made difficult by lack of a consolidated system for reporting or managing data on vessel movements in Australian waters. Some potentially useful data sources on movements of large, registered commercial vessels are:

- The Lloyd’s List Intelligence maintains real-time and archived data on movements of more than 120,000 commercial vessels worldwide. It contains arrival and departure details of all vessels larger than 99 gross tonnes from all major Australian and international ports. The database contains a searchable archive that includes movement histories of boats since December 1997. Searches can be purchased for specific ports, vessels or sequences of vessel movements.

- MarineTraffic provides real-time data on the movements of more than 550,000 vessels. It maintains archived data going back to 2009. Searches can be purchased for specific ports, vessels, areas or periods of time.
- Local port authorities keep records of all vessel movements at their port berths and associated anchorage points.
- The Australian Fisheries Management Authority manages data on the locations of all fishing vessels that have Commonwealth fishing concessions. All Commonwealth fishing concession holders must have installed and be operating an integrated computer vessel monitoring system. The system is also required for some fisheries managed by state and territory fisheries management agencies (such as the Queensland East Coast Trawl Fishery).
- The Bureau of Infrastructure, Transport and Regional Economics maintains statistics on maritime trade, markets, shipping lanes, key trade routes, traded commodities and passenger services throughout Australia.
- The Department of Agriculture and the Australian Border Force maintain data on all vessels arriving in Australian waters from overseas. These data are for proclaimed first ports of entry into Australia.
- The Australian Maritime Safety Authority deals with maritime safety, protection of the marine environment and maritime and aviation search and rescue services. It also coordinates a vessel tracking program, which works as an umbrella for managing related vessel information from the Modernised Australian Ship Tracking and Reporting System (MASTREP) the Great Barrier Reef and Torres Strait Vessel Traffic Service, the Automatic Identification System, the Long Range Information and Tracking system and the Australian Maritime Identification System.
- The aquaculture industry deals with equipment, stock and boat movements between aquaculture sites.

There are no consolidated data on domestic movements of smaller coastal vessels within Australian waters. Ports and some marina operators keep records of vessels that have used their facilities. Local industry groups (such as fishing, petroleum exploration) may provide points of contact for vessels from individual industry sectors that have visited the infested area. Some data may also be available from sources such as the Australian Volunteer Coast Guard, in the form of logged vessel trip reports.

Some states and territories have developed vessel-tracking systems for a range of vessel types. During the operational period of the *Mytilopsis sallei* incursion in Darwin, the Northern Territory Police and the Australian Government Department of Agriculture, with support and input from the Darwin Port Authority, Australian Border Force, the Northern Territory Fisheries Division Licensing Branch, the Australian Fisheries Management Authority and Coastwatch, developed an access database that contained vessel names and contacts, current location, history of individual vessel movements and the risk status of the vessel.

Ocean current modelling

Ocean current modelling may be an effective forward and backward tracing method for estimating the source and sink locations as part of marine pest incursions. There are a number of tools that can assist with modelling of current movements:

Connie3 uses archived currents from oceanographic models and particle tracking techniques to estimate connectivity statistics from user-specified source or sink regions. A range of physical and biological behaviours can be specified including vertical migration, horizontal propulsion, swimming, flotation or surface slick formation.

Regional Ocean Modelling System (ROMS) is an ocean model used for a diverse range of applications. ROMS has pre and post-processing software for data preparation, analysis, plotting and visualisation.

5 Controlling, eradicating and treating established populations

The feasibility of controlling an *Undaria pinnatifida* infestation in Australian waters depends on the nature and location of the incursion and the management strategy adopted. Two control options are available:

- eradication or complete elimination of *U. pinnatifida* from the infested area (highest level of control measure and cost)

  or

- containment and control by limiting the species to the infested area, preventing further spread and protecting uninfected areas (has ongoing costs and implementation so may have higher cost in the long term).

5.1 Eradication

Eradication of *Undaria pinnatifida* requires complete removal from the infested area or destruction. Eradication is unlikely to be successful or feasible if initial investigations determine that the species is widespread, cannot be contained, is difficult to detect, or is present or potentially present in open coastal environments. Eradication is most likely to be feasible when:

- the area inhabited by *U. pinnatifida* is small (less than 1,000 m^2)
- the infestation occurs within an area of minimal flushing or exchange of water
- there is evidence that the population has not reproduced
- the available habitat occurs in relatively shallow waters (less than 5 m)
- the population is relatively aggregated.

See section 6 for treatment options.

5.2 Containment and control

If the decision is made not to attempt eradication but to implement containment and control, the Incident Manager will recommend that interim containment measures be implemented to minimise the risk of pest translocation from the infested waterway. This may include movement controls on potential vectors, public awareness campaigns, policies and practices (in consultation with stakeholders) for vessel and equipment sanitation and surveillance, and control of secondary infestations outside the infested waterway.

National control plans (NCPs) have been developed for several marine pests—including *Undaria pinnatifida*—that are already established in Australia and are having significant impacts on the marine environment or marine industries. The purpose of the NCP is to reflect an agreed national response to reduce impacts and minimise spread of agreed pests of concern.

Each plan includes:

- practical management actions and cost-effective approaches to control or reduce the impact of the marine pest
- recommendations for future research and development, including cost–benefit analysis and planning tools
- links to the National System monitoring strategy
- recommendations for additional public awareness and education strategies
- an implementation strategy.

5.3 Guidelines for delimiting surveys

A delimiting survey establishes the boundary of an area considered to be infested by or free from a pest. The survey should be conducted to establish the area considered to be infested by the pest during the emergency response and to decide if eradication is feasible. The State or Local Control Centre will plan a survey strategy with reference to appropriate confidence limits based on:

- the location where the pest was initially detected
- pest biology—survival, reproductive rate, spread, dispersal and influence of environmental factors
- pest habitat—distribution and suitability of potential habitats around restricted areas and control areas
- survey design—should take into account the sensitivity of the methods to detect the pest species and the ease with which a sample may be obtained, as well as operator safety
- sampling methods—should take into account the area of expected occurrence
- a predictive analysis of areas where the pest is likely to occur
- expected prevalence of the pest if unrestricted
- statistical methods to specify the different confidence limits for targeted and general surveillance.

When possible, the survey should be consistent with national standards and contain estimates of confidence based on best available information.

5.4 Design of a delimiting survey

The location at which the pest was first detected is a useful starting point for a delimiting survey, but it is important to recognise that it is not necessarily the initial site of the infestation. When designing a delimiting survey, it can be useful to work backward, to try to trace the initial source of the incursion (trace-back) and also to try to predict where the pest has, or could, spread to (trace-forward).

The geographic extent of an incursion will be determined by:

- how long the pest has been present at the site before it was detected
- the frequency and quantity of reproductive output from the population since the initial incursion
- the effects of environmental and human factors on the spread of dispersal stages.

Local knowledge and site inspections as well as satellite imagery, hydrographic charts and online databases such as Seamap Australia can be useful for identifying areas that may contain habitat suitable for the pest. Where they exist, hydrodynamic models (for example, CSIRO's Connie3) may also be useful for simulating the likely directions of current flow and the possible rate and extent of spread of planktonic larvae from the known area of infestation. Trace-forward techniques should be used to identify locations outside the infested area that may have been exposed to the pests by vectors that have departed the area known to be infested.

Trace back information can also be used to determine the possible extent of an incursion (particularly a primary incursion where a single size class is present). Working backwards from the estimated age of the specimens and the known settlement biology and larval lifecycle of the species, ocean current modelling can predict the source of a spawning event. This source information can then be used to determine where else in the area the prevailing currents could have spread the larvae.

The greatest survey effort should be made at the margins of the known infestation. Adaptive sampling designs with sample points located on systematic grids or gradients away from the site of known infestation (Eberhardt & Thomas 1991; Gust & Inglis 2006) are most useful to ensure the greatest possible area is covered, while providing the best chance of detecting established and founding populations.

5.4.1 Sampling methods

The type of sampling method chosen should be based specifically on the species being targeted, the habitat being searched and the conditions at the site. *Undaria pinnatifida* occurs predominantly on hard natural and artificial substrata, although it may also foul shelled organisms (such as epifaunal bivalves) that occur in soft-sediment environments. Intertidal habitats should be surveyed visually (from shore or sea) during low tide for occurrences of infestation. In shallow subtidal waters (depth less than 10 m), where *U. pinnatifida* is most abundant, diver or snorkeler visual surveys for the presence of sporophytes are likely to be the most efficient, because a large area can be searched relatively quickly and complex artificial structures (such as wharf pilings, pontoons and niche areas of vessels) can be inspected. ROVs can be used to do this where diving is not an option for safety reasons.

The ability of divers and other forms of visual survey to detect *U. pinnatifida* depends on sufficient training in identification and search techniques, water clarity at the site and the abundance and degree of aggregation of the population. Where underwater visibility is less than 1 m, visual surveys may be severely compromised. Visual searches implemented outside the growing season, when the sporophyte has senesced, may fail to detect any individuals.

Artificial structures such as floating pontoons, projecting piles, steel facings, ropes and mooring dolphins that have large densities of mussels and other fouling biota should be considered a high priority during surveying, as they very susceptible to *U. pinnatifida* fouling. Other surfaces with potential for colonisation include breakwaters, groynes, rock walls, wrecks, hulks, moorings, navigational markers, hulls of moored vessels, aquaculture facilities and natural rocky reefs. In areas where visibility is less than 1 m, visual survey methods will be inefficient (NSPMMPI 2010).

See the Australian marine pest monitoring guidelines, version 2 (NSPMMPI 2010) for additional information that can be adapted for delimiting surveys.

6 Methods for treating established populations

Methods used to treat established populations of *Undaria pinnatifida* will vary in efficacy according to the size and location of the incursion. This chapter summarises treatment options for closed or semi-enclosed coastal environments and for open coastal environments.

6.1 Closed or semi-enclosed coastal environments

Eradication is most achievable in closed or semi-enclosed coastal environments (such as locked marinas and coastal lakes) because the pest can be more easily contained and it is possible to maintain conditions necessary to achieve mortality for longer. Various treatment options are possible in these circumstances, including draining, de-oxygenation and/or flushing of the waterway with fresh water, application of chemical biocides, physical removal and ecological control (Aquenal 2007).

If the infestation is confined to relatively small, enclosed or semi-enclosed waterways, it may be possible to treat the entire water body and all marine habitats within it. If this is not possible, the success of management will depend more heavily on the ability of monitoring and delimitation surveys to locate and treat all clusters of the population. Where resources allow, all habitat potentially suitable for *U. pinnatifida* should be treated. Where this is not possible, habitats should be based on suitability for the pest and delimitation survey results.

6.1.1 Chemical treatments

Major constraints for chemical treatment of water bodies are the volume of water that needs to be treated (a function of the area, depth and degree of flushing of the waterway), the presence and susceptibility of valued non-target organisms that may also be affected, residual effects of any toxicants on the surrounding environment and human health and safety management when handling large volumes of chemicals. Legal issues can also influence the ability to administer chemicals as a rapid response, due to the large number of chemical products available and different legislative requirements between Australian states and territories (Aquenal 2007). Consideration should be given as to whether a permit for the use of chemicals is required from the relevant state or Northern Territory environment agency or the Australian Pesticides and Veterinary Medicine Authority.

6.1.1.1 Chemical options

Macroalgae toxicity has been studied in various species. Copper sulphate, various commercial herbicides, antifoulants, chlorine and lime have been trialled on specific invasive species of macroalgae.

Copper sulphate is known to have a significant effect on plankton mortality, and its persistence in a marine habitat can have serious environmental effects. Copper ions have been used in different conditions and the effect on *Caulerpa taxifolia* has been examined. Laboratory experiments indicate that a copper-ion concentration of greater than 10 ppm (10 mg/L) causes complete *C. taxifolia* mortality after 30 minutes' contact. The effects on *U. pinnatifida* are not as well documented (McEnnulty et al. 2000). The concentration of copper ions required to cause 100% mortality in *C.*

taxifolia was 10,000 times lower than concentrations trialled with sodium ions and potassium; the effects of hypochlorite, produced in situ, were temporary, as recovery occurred after 96 hours (Uchimura et al. 2000).

The effects of various chemical herbicides have been examined on *U. pinnatifida* gametophytes. Mortality has been achieved in laboratory conditions using a commercial antifoulant (Sea-Nine 211) at a concentration of greater than 1.6 mg/L and a red algal extract (furanone 281) at 40 mg/L. The effects of commercial herbicides (atrazine, Diuron, Casuron, Coptrol and various combinations) and other toxicants on *U. pinnatifida* depend on techniques to apply them in the marine environment (Sanderson 1996). Species-specific chemicals have proven difficult to develop and administer for *U. pinnatifida* because of difficulties in applying compounds in the marine environment. Sanderson (1996) trialled several methods of applying herbicides to *U. pinnatifida*, such as injecting it into the plant, applying gel, attaching sponges saturated with chemicals and applying compounds to a bag surrounding the thallus. These methods proved to be costly and labour-intensive and did not achieve a significant impact on populations.

6.1.2 Physical treatments

Physical removal is the most socially and environmentally acceptable way of removing *U. pinnatifida* from a marine system. However, it has been used against *U. pinnatifida* with varying degrees of success (McEnnulty et al. 2000). Manual removal is effective in removing the macroscopic life stages (such as the sporophyte), but recolonisation occurs from microscopic stages (zoospores, gametophytes or sporelings) that cannot be removed by hand. Physical removal of sporophytes is costly, time-consuming and less effective when the incursion is large.

Mature sporophytes can be removed by cutting the plant beneath the sporophyll. Plants must be removed from water and disposed of on land to minimise the chance of release of reproductive material (Sinner et al. 2000b). The preferred period for manual removal of *U. pinnatifida* sporophytes is before zoospore release (Curiel et al. 2001). In Tinderbox Marine Reserve, Tasmania, manual removal was undertaken monthly to prevent the release of additional zoospores (Hewitt et al. 2005). Regular removal of sporophytes (including the holdfasts) throughout the growth season prevented newly recruited sporophytes from reaching maturity and developing sporophylls. Recruitment of new sporophytes can occur from gametophytes or microscopic sporophytes that persist in the benthos or from zoospores released from sporophytes in adjacent areas.

Diver collection and manual removal of *U. pinnatifida* in Tasmania, Victoria and New Zealand have proven effective at reducing population numbers but the only successful eradications of *U. pinnatifida* have been of very small areas of infested artificial structures (Wooton et al. 2004). Recruitment can continue to occur from a seed bank of microscopic gametophytes and dormant germlings. Therefore, physical removal of the visible sporophyte must be sustained over three to four years and/or accompanied by treatment of surrounding habitats to kill microscopic gametophytes and germling sporophytes, to be effective.

U. pinnatifida was successfully eradicated from a submerged sunken trawler in the Chatham Islands, New Zealand, using specially designed heat treatment methods (Wooton et al. 2004). A total of 524 sporophytes were removed by hand from the vessel. The hull was then treated with heated water to kill any remaining microscopic life stages. Plywood boxes with foam seals and containing electrical elements were attached to the hull using magnets. The elements inside the boxes heated the enclosed seawater to 70 °C. A diesel-generated support vessel maintained heat treatment for 10 minutes to compensate for heat loss, and a petrogen flame torch was used where the curve of the ship hull did not allow the heat box to attach firmly (Wooton et al. 2004). Heat treatment was undertaken on the hull of the ship until all sporophytes were removed and the zone of the ship containing the sporophytes had been treated. Final inspection of the hull, three years after heat treatment, showed that the eradication was successful and no neighbouring areas had been fouled with *U. pinnatifida* (Wooton et al. 2004).

Mechanical removal of *U. pinnatifida* using crusher boats, weed cutters, rotovators or harvesters is unlikely to be successful because it occurs in physically complex habitats and zoospores can be released during sporophyte removal (Aquenal 2007). Removal strategies need to take into consideration long-term viability of sporophylls and potential for seed banks to develop

6.1.2.1 Removable structures

Ropes, mooring lines, buoys, floating pontoons and other structures within the infested area that can be removed from the water should be removed and treated on land. Procedures for treating these structures are described in section 4.1.3.3 and could include:

- disposal to landfill
- air-drying for a minimum of seven days
- high-pressure water blasting
- immersion in chemical or fresh water baths.

6.1.2.2 Hard substrata and structures that cannot be removed from the water

Hard substrata and structures that cannot be removed from the water include intertidal and submerged habitats.

Intertidal habitats

Hard intertidal substrata, such as wharf piles, exposed jetties and rocky shorelines may be treated when they are exposed at low tide.

Submerged habitats

Many traditional methods used for removing mobile species are not appropriate for macroalgae. For submerged habitats, the most commonly used treatment has been manual removal, but slow removal rates limit this method for large-scale efforts. In Bluff, New Zealand, a combination of manual removal and shading of infested areas with black PVC plastic (encapsulation) has resulted in moderate control (but not eradication) of *U. pinnatifida*.

Wrapping and encapsulating submerged substrata using impermeable barriers such as polyethylene plastic have successfully treated fouling on wharf piles, jetties, pontoons, vessel moorings, small reefs and aquaculture facilities, which cannot be removed from the water (Aquenal 2007). Black polyethylene plastic bale wrap (1 m wide and 50 µm thick) is wrapped over the structures, with an overlap of approximately 0.4 m on each successive layer of wrap, and secured using PVC tape to achieve a watertight seal. Aquenal (2007) provides procedures for deploying the wrap on different structures and details on the costs involved with this treatment technique. Wrappings can remain in place for up to 12 months, providing some protection from reinfection. Should the outside of wrappings become reinfested, removing the wrapping provides a second treatment option if the macroalgae can be retained. Commercial divers can treat areas of the structures that cannot be wrapped effectively using an underwater flame torch or steam sterilisation.

Encapsulation techniques are most suited to treating small-sized to medium-sized incursions (less than 10,000 m^2) in relatively sheltered waters. The procedure is very labour intensive and hazards are associated with its deployment by divers. The wrap is susceptible to puncture and tear by shipping, strong water currents and sharp oysters or tubeworms, which reduces its effectiveness. The technique is non-selective and all organisms contained within the wrapping will be killed.

Encapsulation or other containment techniques may also be used in combination with chemical treatment to achieve faster kill rates. Chemicals are injected into the covered area to maintain elevated concentrations of the biocide in close proximity to the fouled surface (Aquenal 2007). For *U. pinnatifida*, this could include injection of sodium hypochlorite or commercial herbicides such as atrazine, Diuron, Casuron or Coptrol.

In New Zealand, the Ministry of Primary Industries, Biosecurity NZ and the Department of Conservation trialled a steam sterilisation unit for killing the microscopic stages of *U. pinnatifida* on submerged reefs and artificial structures. The unit generates hot water or steam at the surface, which is then delivered underwater by a hose, where it sterilises the seabed by heating seawater encapsulated inside a silicone cone (diameter 300 mm, volume 0.004 m^3) that is held against the substrate under treatment (Aquenal 2007). The apparatus can be operated from a support vessel or from land. Fresh water is fed into the unit, either from mains supply or with a water pump, and heated by a continuous-flow, diesel-fuelled water heater (califont), which heats water to 98 °C or generates steam if required. Steam is discharged into the cone until lethal temperatures are attained, and then the unit is repositioned on an adjacent substrate to repeat the treatment process.

Preliminary trials show that, although the technique causes high mortality of *U. pinnatifida*, it is difficult to maintain a sufficiently tight seal of the silicone cone over complex substrata, so not all plants are killed in a single application. Health and safety issues (such as hypothermia and decompression illness) and supply of consumables (such as fuel and water) further limit the effective operation of the sterilisation tool. Effective and safe deployment is likely to be limited to depths no greater than 30 m, but only small areas can be treated without multiple dive teams, even at relatively shallow depths. The total treatable area by a single team of divers over one working day at depths of up to 10 m was approximately six m^2. The amount of treated substrate could, however, be increased by using several dive teams but this would increase costs. Treatment at depths greater than 30 m may require a compression chamber on site and would be limited to the most experienced commercial operators (Aquenal).

6.1.2.3 Soft sediment habitats

Many traditional methods used for removing mobile species are not appropriate for macroalgae. For submerged habitats, the most commonly used treatment has been manual removal, but slow removal rates limit this method for large-scale efforts. In Bluff, New Zealand, a combination of manual removal and shading of infested areas with black PVC plastic (encapsulation) has resulted in moderate control (but not eradication) of *U. pinnatifida*.

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Preliminary trials show that, although the technique causes high mortality of *U. pinnatifida*, it is difficult to maintain a sufficiently tight seal of the silicone cone over complex substrata, so not all plants are killed in a single application. Health and safety issues (such as hypothermia and decompression illness) and supply of consumables (such as fuel and water) further limit the effective operation of the sterilisation tool. Effective and safe deployment is likely to be limited to depths no greater than 30 m, but only small areas can be treated without multiple dive teams, even at relatively shallow depths. The total treatable area by a single team of divers over one working day at depths of up to 10 m was approximately 6 m^2. The amount of treated substrate could, however, be increased by using several dive teams but this would increase costs. Treatment at depths greater than 30 m may require a compression chamber on site and would be limited to the most experienced commercial operators (Aquenal).

6.2 Open coastal environments

Limited emergency eradication response options are available to deal with marine pest incursions occurring in open coastal environments, particularly on high-energy coastlines or in deep water (more than 10 m). Many treatment options described in section 6.1 may be applied to small-scale incursions in these environments, but the main difficulties occur in containing the larvae and maintaining treatment conditions in a lethal state for sufficient time. The latter requires deployment of structures or application technologies that allow delivery of chemicals or encapsulation techniques over large areas and which are robust to water movement.

Successful eradication of small incursions may be possible using simple methods (such as manual removal, smothering, small-scale containment and chemical treatment) if the incursion is detected early or where site-specific conditions allow algae containment and treatment. Trials of steam sterilisation units on subtidal rocky reefs have shown some effectiveness for treating relatively small areas of habitat, but the efficacy of this technique is compromised in complex topographical environments.

6.3 Monitoring and ongoing surveillance

Monitoring and surveillance are used to detect new populations *Undaria pinnatifida* and to inform the eradication and control programs. Active surveillance for the presence of *U. pinnatifida* in restricted and control areas should continue until the incursion is declared eradicated or until the emergency response is stood down. If a zoning program is implemented, it will be necessary to implement targeted active surveillance for the species outside the restricted and control areas to support the declaration of zones free from the algae.

Monitoring Design Package (Version 1c), including the Australian marine pest monitoring manual and guidelines, can be used to help determine appropriate sampling intensity for ongoing surveillance.

Several methods may be appropriate for surveillance:

- systematic, targeted searches by divers or ROVs of suitable or treated sub-tidal habitat within the restricted area or at sites at risk of infection
- systematic, targeted searches by shoreline observers of suitable or treated intertidal habitat within the restricted area or at sites at risk of infection
- targeted searches and inspection of vessels and other vectors departing, or which have left, the control area
- regular monitoring of settlement within the restricted area or at sites at risk of infection.

Appendix A: Guidelines for using the Biosecurity Act during an emergency response to a marine pest of national significance

The following is an interim process for using the Biosecurity Act for action on vessels to treat contaminations by a marine pest of national significance. The Biosecurity Act may be used in certain circumstances, including where a biosecurity officer suspects on reasonable grounds, that the level of biosecurity risk associated with the vessel is unacceptable. Under these circumstances, a biosecurity officer may, in relation to a vessel that is under biosecurity control direct:

- the person in charge or operator of a vessel not to move, interfere with or deal with the vessel
- the person in charge or operator of a vessel to move the vessel to a specified place, including a place outside of Australian territory
- a vessel to undergo treatment action deemed necessary by the biosecurity officer
- that other biosecurity measures which may be prescribed by regulations be undertaken.

In addition, biosecurity officers may exercise certain powers, such as taking samples of ballast water from vessels, for the purpose of monitoring compliance with provisions for the management of ballast water at a port or offshore terminal within the outer limits of the EEZ of Australia. Where the Director of Biosecurity (or delegate) is satisfied that a sample of the vessel's ballast water indicates that the vessel poses an unacceptable level of biosecurity risk, then the Director may give a direction to the vessel not to discharge ballast water until conditions specified in the direction are met.

The conditions of using the Biosecurity Act are:

- The Australian Government Department of Agriculture is to be contacted before taking the proposed action to determine the appropriate provisions of the Biosecurity Act that apply.
- Directions to take action under the Biosecurity Act are to be given by a biosecurity officer. Officers of a state or territory government must be authorised as biosecurity officers under the Biosecurity Act to be able to give directions under the Act.
- Actions under the Biosecurity Act should only be taken for vessels currently identified as at risk of spreading a marine pest of national significance.

Responsibility for directing and approving action under the Biosecurity Act rests with the biosecurity officer, but the actual vessel control and treatment actions are handled by the Local or State Control Centre. As a matter of policy, the following information should be provided to the Australian Government Department of Agriculture to help determine appropriate application of the Biosecurity Act:

- the proposed course of action

- the location of proposed action
- details to identify the vessel involved in the proposed action
- contact details of local management agencies that will be managing the vessel control and treatment.

Appendix B: State and territory legislative powers of intervention and enforcement

The Intergovernmental Agreement on Biosecurity (IGAB), is an agreement between the Australian, state and territory governments. It came into effect in January 2019 and replaced the previous IGAB which started in 2012. The agreement was developed to improve the national biosecurity system by identifying the roles and responsibilities of governments and outlining the priority areas for collaboration to minimise the impact of pests and disease on Australia's economy, environment and community. The National Environmental Biosecurity Response Agreement was the first deliverable of the IGAB and sets out emergency response arrangements, including cost-sharing arrangements, for responding to biosecurity incidents primarily affecting the environment and/or social amenity and when the response is for the public good. In combination with the IGAB, Commonwealth, state and territory governments are responsible under their principle fisheries management legislation to respond consistently and cost-effectively to a marine pest incursion.

Table B1 Commonwealth, state and territory legislation covering emergency response arrangements

Jurisdiction	Agency	Principle acts covering emergency response arrangements	Marine pest contact website
Commonwealth	Department of Agriculture and Water Resources Department of Agriculture	*Fisheries Management Act 1991* *Biosecurity Act 2015*	agriculture.gov.au/fisheries
New South Wales	NSW Department of Primary Industries	*Fisheries Management Biosecurity Act 1995* Fisheries Management (General)Biosecurity Regulation 2017 Fisheries Management (Aquaculture) Regulation 2012 *Ports and Maritime Administration Act 1995* *Marine Parks Regulation 1997* *Marine Safety Act 1998*	dpi.nsw.gov.au/fishing/pests-diseases
Victoria	Victorian Fisheries Authority; Department of Jobs, Precincts and Regions (Agriculture Victoria)	*Fisheries Act 1995* (protection of fisheries) *Environment Protection Act 1970* (management of ballast water) Marine and Coastal Act 2018	https://vfa.vic.gov.au/operational-policy/pests-and-diseases/noxious-aquatic-species-in-victoria/aquatic-pests

Jurisdiction	**Agency**	**Principle acts covering emergency response arrangements**	**Marine pest contact website**
		Marine Safety Act 2010 (power of Harbour Masters to direct vessels and duty of harbour masters to minimise adverse impacts on environment) *Port Management Act 1995* (where no harbour master appointed, powers to direct vessels and act to minimise adverse effects on the environment)	
Queensland	Department of Agriculture and Fisheries	*Fisheries Act 1994* *Biosecurity Act 2014*	daff.qld.gov.au/fisheries/ www.qld.gov.au/environment/coasts-waterways/marine-pests
South Australia	Primary Industries and Regions SA	*Fisheries Management Act 2007*	pir.sa.gov.au/biosecurity/aquatics
Western Australia	Department of Fisheries	*Fish Resources Management Act 1994* (under review)	fish.wa.gov.au/Sustainability-and-Environment/Aquatic-Biosecurity/Pages/default.aspx
Tasmania	Department of Primary Industries, Parks, Water and Environment	*Living Marine Resources Management Act 1995*	dpipwe.tas.gov.au/biosecurity-tasmania/aquatic-pests-and-diseases
Northern Territory	NT Department of Primary Industry and Resources	*Fisheries Act 1988*	nt.gov.au/marine/for-all-harbour-and-boat-users/biosecurity/aquatic-pests-marine-and-freshwater nt.gov.au/d/Fisheries/index.cfm?header=Aquatic%20Biosecurity

Glossary

Term	Definition
CCIMPE	Consultative Committee on Introduced Marine Pest Emergencies
DSE	Department of Environment and Primary industries (Victoria)
EMPPlan	Emergency Marine Pest Plan
IGAB	Intergovernmental Agreement on Biosecurity
IMO	International Maritime Organization
NBIRP	National biosecurity incident response plan
NEBRA	National Environmental Biosecurity Response Agreement
NIMPIS	National Introduced Marine Pest Information System
RRM	Rapid response manuals

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