Control of alligator weed with herbicides: A review

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Introduction

Alligator weed (*Alternanthera philoxeroides* (Mart.) Griseb.) is a widespread weed that is difficult to control. Despite many published accounts relating to its control there is no published review on herbicide control of alligator weed. This paper describes the biology and impacts of alligator weed and the efficacy of herbicides and herbicide-based control programs used against it.

Global naturalisation

Alligator weed is native to the Parana River area of South America (Julien *et al.* 1995). It has subsequently spread to many countries outside of its indigenous range. It was first detected in the United States of America (USA) in 1897, in Mobile, Alabama, growing in ballast presumably sourced from its native range (Zeiger 1967). It was also collected from ballast in 1906, at Aratapu (Northland) in New Zealand (Cheeseman 1906). It was introduced as a forage crop to China in the 1930's (Wang *et al*. 2005) and had established in India, Burma and Indonesia by the 1960's (Sculthorpe 1967). It was first detected in Australia at Newcastle in 1946 and is likely to have been introduced with cargo during World War II (Julien and Bourne 1988). More recent introductions have occurred to Puerto Rico, Singapore, Vietnam and Thailand (Julien and Broadbent 1980). It was first found in Sri Lanka in 1999, established as a food plant possibly introduced from Australia (Tegan 2009), Italy (Garbari and Pedulla 2001) and France in 2002 (EPPO 2012).

Biology

Alligator weed is a perennial stoloniferous herb in the family Amaranthaceae. It requires a warm growing season but can tolerate a wide range of climate conditions, including winter frosts, which will kill exposed stem material. Although it flowers producing peduncled capitate inflorescences, seeds are not produced in its introduced range and it reproduces solely by clonal growth (Gangstad 1978, Julien and Broadbent 1980, Julien *et al*. 1992).

Alligator weed is an amphibious plant. It grows as an aquatic plant, either rooted into the hydrosoil and emerging above the water, or as a sprawling mat of entangled stems floating over the water, or in ephemerally damp or swampy soil (Julien and Broadbent 1980). It can also establish and become weedy in terrestrial habitats, including pasture (Julien and Broadbent 1980), urban areas (Gunasekera and Bonilla 2001) and arable crops (Shen *et al*. 2005).

Dispersal of alligator weed in water is aided by its hollow stems allowing dislodged stem fragments to float in water and drift with currents to lodge at new locations (Eggler 1953, Julien and Broadbent 1980, Dugdale *et al*. 2010). Any stem or taproot fragment that contains a bud, which breaks off the plant is capable of regeneration. Gangstad (1978) also reports that ~5% of internodes produce roots when floating in water but not when on soil. Julien *et al*. (1992) estimated that 9,800 to 17,300 stem nodes m^{-2} yr^{-1} were produced by alligator weed, representing its reproductive potential. It can also be spread by physical transport of stem or root fragments on machinery, boats, fishing nets or animals e.g. trapped within the cloven hoofs of livestock.

Alligator weed is also dispersed deliberately by humans. In Australia, it was cultivated as a food plant in home gardens by members of the Sri Lankan community in the mistaken belief that it was mukunuwenna or sessile joy weed (*Alternanthera sessilis* (L.) R.Br. ex DC.), which is very popular in Sri Lanka. In 2001, it was cultivated in at least 760 home gardens and there were 13 known naturalised sites in Victoria. However, deliberate cultivation in Australia has been reduced because of a very successful education campaign and provision of an alternative native plant (*Alternanthera denticulata* R.Br., Gunasekera and Bonilla 2001).

Alligator weed is more vigorous in aquatic situations, where it has taller, thicker stems and large amounts of above-soil biomass than when it is growing terrestrially (Julien and Broadbent 1980, Julien *et al*. 1992). Floating mats become very dense and have reached ~2 kg DW m^{-2} in Louisiana, USA (Lapham 1964). An aquatic infestation in Melbourne, Australia, spread outwards from bankside patches to form floating mats that increased in area by 45 to 205% per annum over five years. The average biomass at the site was 4.3 kg DW m^{-2}, consisting of emergent and submerged biomass (Clements *et al*. 2011). For terrestrial alligator weed, Julien and Bourne (1988) report an annual increase in biomass per unit area of 22% as it gradually displaced competing pasture species. Over 7.3 kg DW m^{-2} has been collected from a terrestrial site where it had been present for 20 years, with 10 fold higher biomass below ground than above (Schooler *et al*. 2008).

Alligator weed exhibits phenotypic plasticity with different growth forms occurring in aquatic and terrestrial environments, which are not driven by genetic differences (Li and Ye 2006, Geng *et al*. 2007). Terrestrial plants have a tough, fleshy, tap-root like structure that is lacking in the aquatic plants; their stems are thinner and more lignified; they have smaller internodal cavities; have shorter internodes; have less leaf area per stem; less relative chlorophyll content; and have lower growth rates than alligator weed growing at aquatic sites (Julien *et al*. 1992, Geng *et al*. 2007). Terrestrial plants also have a much greater biomass of root material than aquatic alligator weed (Julien and Broadbent 1980, Julien *et al*. 1992, Geng *et al*. 2007). This phenotypic plasticity is an important adaptive strategy for alligator weed because it allows a single invading genotype to adapt to a wide range of habitats in introduced areas, and therefore increases the chance of proliferation. Furthermore, Wilson *et al*. (2007) have shown that alligator weed develops a different morphology after physical control. Plants that were subject to shoot removal, just above soil level to mimic mowing, had higher below ground root biomass, a higher ratio of root to stem biomass and positioned its leaves much closer to the ground. In addition to this phenotypic plasticity, where genetically identical plants have different growth forms depending on their habitat, alligator weed may also have different growth forms that are due to a genetic basis (biotypes) (Kay and Haller 1982).

Differences in morphology or biotype are important because herbicide control can be affected. For example, Kay (1992) found that a slender stem biotype was more susceptible to quinclorac herbicide than a broad stem biotype. A difference in susceptibility of the terrestrial and aquatic forms of alligator weed to biological control has also been shown, where effective control of the aquatic form is achieved but not for the terrestrial form (Spencer and Coulson 1976, Julien and Broadbent

1980, Kay and Haller 1982, Li and Ye 2006). Such differences may be an additional cause of observed variable responses of alligator weed to herbicide treatment, which have been attributed to differences in water level and temperature (Steenis and McGilvery 1961).

Impacts

Alligator weed grows rapidly over the surface of water bodies forming floating mats that cover the water surface, impeding access, navigation, water-use and drainage (Spencer and Coulson 1976, Gangstad 1978, Julien and Broadbent 1980, Clements *et al*. 2011). It alters the timing and magnitude of litter inputs into water along with having a faster decomposition rate than native riparian communities (Bassett *et al*. 2010). It has also been shown to reduce native invertebrate densities and alter community composition (Pan *et al*. 2010) and its evapotranspiration increases water loss from standing water relative to water without vegetation (Boyd 1987).

It displaces and competes with native plant communities, and pasture and cropping species in moist agricultural areas, including rice (*Oryza sativa* L.). It provides mosquito habitat, restricts light penetration into the underlying water and prevents gas exchange between the water and atmosphere (Gangstad 1978, Julien and Broadbent 1980, Julien and Bourne 1988, Shen *et al*. 2005). It is palatable to livestock, but the grazing of alligator weed has been associated with photosensitivity and resultant skin lesions, liver damage and death in cattle, calves and lambs (Bourke and Rayward 2003). Farming practices for cattle are affected by alligator weed in Kaipara District, New Zealand. Young livestock are initially raised in hill country, away from areas where this species occurs and black cattle are favoured (as white patches are most sensitive to damage). Crops such as kumara (*Ipomoea batatas* (L.) Lam.) and rice can be heavily impacted by alligator weed. Yield losses range up to 45% for rice (Shen *et al*. 2005) and some Northland kumara crops are lost when heavy infestations of alligator weed occur (P. Joynt, Northland Regional Council personal communication).

These impacts have led to alligator weed being regarded as one of the world's worst weeds (Holm *et al*. 1997) and it is a legislated weed in most countries where it occurs. In Australia, it is one of the original 20 Weeds of National Significance (WoNS), which are managed nationally (van Oosterhout 2007). In New Zealand, it is designated an unwanted organism and is actively managed at all known sites outside of the Northland Region (Champion 2008). In China, US$72 million is spent each year to manage it (Liu and Diamond 2005) and there are occasional eradication efforts at local scales (Pan *et al*. 2010). In USA, there has been considerable effort to control it since the 1950's (Gangstad 1978).

Efficacy of herbicides on alligator weed

Many publications exist that describe use of a large number of herbicides on alligator weed in various control programs, however much of the data presented are inadequate to allow efficacy of the herbicide to be determined and comparisons made. Sixteen publications were found that report on the efficacy of 32 herbicides in 49 combinations on alligator weed (Table 1). Short- (4 to 16 weeks), medium- (18 to 38 weeks), and long-term (~52 weeks) efficacy of each of the herbicides tested in these publications are shown (Table 1).

Although several herbicides achieved good to excellent control (80–89% and 90–100%, respectively) in the short- and medium-term, good to excellent long-term control was not as common and usually required repeated herbicide applications. For example, good to excellent long-term control was reported on eight occasions where herbicide was applied more than once: glyphosate three or four times within 12 months (Schooler *et al*. 2008); imazapyr once per year for two years (Langeland 1986a) and twice in 10 months (Hofstra and Champion 2010); metsulfuron-methyl, three or four times within 12 months (Schooler *et al*. 2008), five times over two years (Schooler *et al*. 2010) and twice in 10 months (Hofstra and Champion 2010); and triclopyr, three or four times within 12 months (Schooler *et al*. 2008) and twice in 10 months (Hofstra and Champion 2010).

Where herbicide was only applied once, good to excellent long-term control was only achieved on five occasions, twice with imazapyr (Allen *et al*. 2007, Hofstra and Champion 2010) and once each with dichlobenil, karbutilate (Blackburn and Durden 1974), and metsulfuron-methyl (Hofstra and Champion 2010). However, only a single application of metsulfuron-methyl and imazapyr to young plants (3-months old) resulted in excellent control, with older plants being less effected (Hofstra and Champion 2010). For established plants, Allen *et al*. (2007) reported that imazapyr was applied once to alligator weed in the spring and autumn, and only the autumn application resulted in effective long-term control. They suggested that this was because of better downward translocation of the herbicide at this time of year. The marshes that they did the experiment in were inundated after herbicide application, which offers an alternative explanation for the effective control after a single application because inundation after herbicide application has been observed anecdotally to improve control with herbicides (Steenis and McGilvery 1961, Langeland 1986a). Although the mechanism for this improved control is not known, it seems likely that alligator weed under water will be stressed through reduced gas exchange capacity and reduced light availability.

The need for multiple herbicide applications was also demonstrated by Bowmer *et al*. (1991), who tested at least 100 combinations of herbicides in ten experiments in replicated damp terrestrial plots over three years in New South Wales, Australia. They found that no single herbicide application eradicated alligator weed; repeat applications were always necessary. The best treatment regimes were 1) a single application of dichlobenil followed by metsulfuron or glyphosate nine months later or 2) three applications of metsulfuron or metsulfuron + glyphosate over 18 months.

Relative effectiveness

The relative effectiveness of herbicides that achieved good to excellent control has been examined by a number of authors, where a selection of the herbicides glyphosate, imazapyr, metsulfuron-methyl and triclopyr triethylamine (TEA) have been tested against each other in long-term studies. Imazapyr and metsulfuron-methyl provide the best and most consistent control of alligator weed while triclopyr TEA and glyphosate are less effective.

Imazapyr and triclopyr amine were tested against alligator weed by Allen *et al*. (2007) in managed marshes in Alabama and Georgia, USA. Either herbicide was applied to 5 m by 5 m plots at one of three rates within the range recommended by the manufacturer in April (spring) or July (summer). In the autumn following application, both herbicides achieved almost 90% biomass reduction relative to controls when applied in summer and there was no difference in biomass with application rate. Only the high rate triclopyr and mid and high rate imazapyr still had significantly less biomass than controls by the second autumn after summer application (78, 88 and 99%, respectively). For the spring application, only the high rate of imazapyr resulted in a statistically significant biomass reduction by the first autumn (84%, relative to controls) and none of the treatments resulted in control by the second autumn after application.

Hofstra and Champion (2010) treated plants grown outdoors rooted in tanks with a single application of imazapyr, metsulfuron-methyl, triclopyr TEA or triclopyr TEA + picloram. For young plants there was regrowth observed for all treatments, beginning at 2.5 MAT (months after treatment) depending on treatment, but this was not substantial for imazapyr and triclopyr + picloram until 10-11 MAT (the spring/summer following summer application). Despite this apparent difference between treatments, reductions in biomass did not differ greatly between any of the treatments one year after application (ranging from a 75 to 90% reduction compared to controls). Two applications to mature plants of metsulfuron-methyl and imazapyr resulted in a 75 to 80% reduction in biomass, relative to controls, while biomass was not reduced for glyphosate and triclopyr TEA. Single applications of all four herbicides did not reduce biomass, relative to controls.

Schooler *et al.* (2008) investigated the relative effectiveness of broad spectrum (glyphosate) and selective (metsulfuron-methyl and triclopyr TEA) herbicides in moist terrestrial situations. Each herbicide was applied at two different frequency regimes (December, January, March and June or December, January and March). There was no difference in efficacy between metsulfuron-methyl and triclopyr TEA. Although, in the long-term there was no difference between the four- and three-application regimes, the broad spectrum herbicide reduced below ground biomass by ~80%, while the selective herbicides reduced below ground biomass by 95%. Above ground biomass was reduced by ~90% by the selective herbicides but not reduced by the broad spectrum herbicide, relative to the control. An important finding of this study is that even the poorest performing herbicide regime (3 applications of glyphosate) that was tested, which resulted in above ground biomass no different to controls, resulted in an 81% reduction of below ground biomass, 15 months after herbicide application. Reduction in alligator weed below ground biomass after herbicide application has also been demonstrated by Schooler *et al.* (2007) and Hofstra and Champion (2010).

The studies of Hofstra and Champion (2010) and Schooler *et al.* (2008) corroborate the recommendations of Champion (2008), van Oosterhout (2007), Sainty *et al.* (1998) and Bowmer *et al.* (1991) who all recommend multiple annual metsulfuron-methyl treatments of alligator weed in New Zealand and Australia. Hofstra and Champion (2010) found that the second application of glyphosate did not provide improved control of alligator weed; again this corroborates field experience in New Zealand (Champion 2002) and Australia (van Oosterhout 2007), where glyphosate does not provide adequate control in the long-term for rooted alligator weed.

The findings of Schooler *et al.* (2008) provide credibility to eradication programs in Australia and New Zealand that rely on a resource depletion strategy, whereby the below ground portions are gradually depleted with successive herbicide applications, ultimately aiming to eradicate the plant (see Control Programs). They also suggest that, despite the renowned poor translocation of herbicides to below ground parts of alligator weed (see Translocation of Herbicides), herbicide regimes can still be effective in reducing below ground biomass. Whether this is due to damage to the roots by the phytotoxicity of the herbicide itself or depletion of carbohydrates as the plant repeatedly re-establishes shoots and leaves is unknown. They also demonstrate the importance of determining below ground biomass, which is a key carbohydrate reserve and source of regenerating propagules. The importance of reducing below ground material was recognised by Bowmer *et al.* (1991) who determined the reductions in below ground biomass two months after treatment with sulfometuron-methyl, dichlobenil, glyphosate, glyphosate + clopyralid and imazapyr was 96, 91, 80, 29 and 16%, respectively, relative to controls. Remaining roots were also collected, cut at the internodes and then grown in potting mix. They found that all treatments reduced the viability of the underground biomass by 99, 94, 89, 88 and 78% relative to controls, respectively (which had 30,412 fragments m^{-2}). These studies provide support for the gradual depletion approach to controlling or eradicating alligator weed. Given no other field studies have reported the impact on below ground biomass in relation to herbicide application and only two mesocosm studies do (Hofstra and Champion 2010, Schooler *et al.* 2007), we suggest more research would be valuable in determining the effectiveness of control programs in reducing below ground biomass.

Another factor that impacts on the usefulness of herbicides against alligator weed is stem fragmentation. Recent studies have confirmed that herbicide application creates viable alligator weed stem fragments (Dugdale *et al.* 2010, Clements *et al.* 2012). In aquatic situations, these fragments have the potential to disperse and establish new infestations. Approximately 40% of stem fragments produced after application of metsulfuron-methyl were viable compared to up to 2% after the application of glyphosate (Dugdale *et al.* 2010, Clements *et al.* 2012). Given each of these fragments has the potential to create a new infestation at an unknown downstream location, frequent applications of glyphosate on aquatic infestations may provide a better outcome than metsulfuron, in containment of infestations within catchments, even though the later kills the initial patch more effectively.

Translocation of herbicides

At least eight studies have been conducted on the absorption and translocation of radio-labelled herbicides in alligator weed (Earle *et al.* 1951, Funderburk and Lawrence 1963, Gangstad 1978, Bowmer *et al.* 1991, Bowmer and Eberbach 1993, Bowmer *et al.* 1993, Tucker *et al.* 1994, Eberbach and Bowmer 1995). These studies have demonstrated that herbicide translocation is poor within alligator weed, providing insight into why herbicide control of alligator weed is difficult with foliar applied herbicides.

The first step of herbicide translocation is absorption into the leaves. For alligator weed, absorption occurs at rates similar to other species for glyphosate and imazapyr (Tucker *et al.* 1994). In one study, 25 to 41% of foliar-applied glyphosate absorbed into alligator weed leaves (depending on spray droplet size, Bowmer *et al.* 1993), while in another study 42% was absorbed (Tucker *et al.* 1994). Absorption of imazapyr was much higher at 88% (Tucker *et al.* 1994). This occurs in alligator weed at a rate similar to other species for glyphosate and imazapyr (Tucker *et al.* 1994), with 25 to 41% of applied glyphosate absorbed into leaves (depending on spray droplet size, Bowmer *et al.* 1993) and 42 and 88% of glyphosate and imazapyr, respectively (Tucker *et al.* 1994). However, subsequent translocation out of the leaves to other plant parts is limited, with values ranging from 5 to 20% (Bowmer and Eberbach 1993, Bowmer *et al.* 1993).

Only 15% of applied imazapyr was translocated to the roots (Tucker *et al.* 1994), while translocation of glyphosate to roots has been reported as 0.9% (Tucker *et al.* 1994), 7% (Bowmer *et al.* 1993) and 1.6 to 3.3% (Bowmer and Eberbach 1993). This poor translocation results in low concentrations of herbicide within the roots, which can be below the phytotoxic threshold (Bowmer *et al.* 1993). Imazapyr is phytotoxic at lower doses than glyphosate (Tucker *et al.* 1994), and this combined with the increased translocation of imazapyr to roots and underground storage tissues may be the mechanism responsible for the observed susceptibility of alligator weed to imazapyr relative to glyphosate.

Given that such small amounts of herbicide reach the below ground plant parts, plant size can have a large effect on the concentration that is achieved. Bowmer and Eberbach (1993) showed that the concentration of glyphosate in tissues was smaller in large plants relative to small plants, resulting in glyphosate values below phytotoxic levels for large plants. This data suggests that although large plants provide lots of foliage for optimal herbicide interception, this benefit could be outweighed by the subsequent dilution of the adsorbed glyphosate within the plant tissues.

The results above for alligator weed are unusual, particularly for glyphosate, which in most plants is rapidly translocated from leaves to metabolic sinks, especially meristematic and storage

Table 1. Short-, medium-, and long-term efficacy of herbicides on alligator weed.

Herbicide	No. of studies	Short term control ~4-16 WAT	Medium term control ~18-38 WAT	Long term control ~52 WAT
2,4,5-T (PGBEE)	4	**Excellent** (Blackburn 1963; 22.4 kg ha^{-1}; floating aquatic, greenhouse) **Good** (Lapham 1964; 8.97 kg a.e. ha^{-1}; floating aquatic, field@: Gangstad *et al.* 1975; 8.4 kg ha^{-1}; aquatic, field) **Fair** (Blackburn 1963; 5.6 kg ha^{-1}; floating aquatic, greenhouse): Poor (Gangstad *et al.* 1975; 5 and 2.7 kg ha^{-1}; aquatic, field)	**Poor** (Spencer 1968; 4.5 kg a.e. ha^{-1}; rooted aquatic, field)	**Poor** (Spencer 1968; 4.5 kg a.e. ha^{-1}; rooted aquatic, field)
2,4,5-T (PGBEE) + amitrole	2	**Excellent** (Spencer 1968; 13.5 + 3.4 kg a.e. ha^{-1}; rooted aquatic, field)	**Fair** (Spencer 1968; 13.5 + 3.4 kg a.e. ha^{-1}; rooted aquatic, field) **Poor** (Bowmer *et al.* 1991; rate not stated; terrestrial, field)	**Poor** (Spencer 1968; 13.5 + 3.4 kg a.e. ha^{-1}; rooted aquatic, field)
2,4,5-T (PGBEE) + amitrole + picloram	1	**Excellent** (Spencer 1968; 22.4 + 1.1 + 2.2 kg a.e. ha^{-1}; rooted aquatic, field)	**Poor** (Spencer 1968; 22.4 + 1.1 + 2.2 kg a.e. ha^{-1}; rooted aquatic, field)	**Poor** (Spencer 1968; 22.4 + 1.1 + 2.2 kg a.e. ha^{-1}; rooted aquatic, field)
2,4,5-T (PGBEE) + dichlobenil (liquid)	1	**Excellent** (Lapham 1964; sequential application, 4.5 + 6.7 kg a.e. ha^{-1}; floating aquatic, field@) **Fair** (Lapham 1964, as mixture; 4.5 + 6.7 kg a.e. ha^{-1}; floating aquatic, field@)		
2,4-D (dode-cylamine)	3	**Good** (Langeland 1986a; 3.2 g a.e. L^{-1}; rooted aquatic, field) **Poor** (Lapham 1964; 2.8 kg a.e. ha^{-1}; floating aquatic, field@)	**Poor** (Langeland 1986a; 3.2 g a.e. L^{-1}; rooted aquatic, field: Bowmer *et al.* 1991; rate not stated; terrestrial, field)	
2,4-D (amine) + dichl-obenil (liquid)	1	**Excellent** (Lapham 1964; sequential application, 2.8 + 6.7 kg a.e. ha^{-1}; floating aquatic, field@) **Fair** (Lapham 1964; as mixture, 2.8 + 6.7 kg a.e. ha^{-1}; floating aquatic, field@)		
2,4-D (dode-cylamine) + dicamba (dimethyl-amine)	1	**Excellent** (Langeland 1986a; 1.2 + 2.3 g a.e. L^{-1}; rooted aquatic, field)	**Poor** (Langeland 1986a; 1.2 + 2.3 g a.e. L^{-1}; rooted aquatic, field)	
2,4-D (PGBEE)	5	**Excellent** (Gangstad *et al.* 1975; 8.4 and 5 kg ha^{-1}; aquatic, field) **Fair** (Blackburn 1963; 22.4 kg ha^{-1}; floating aquatic, greenhouse: Gangstad *et al.* 1975; 2.7 kg ha^{-1}, aquatic, field) **Poor** (Blackburn 1963; 5.6 kg ha^{-1}; floating aquatic, greenhouse: Lapham 1964; 9 kg a.e. ha^{-1}; floating aquatic, field@)	**Poor** (Spencer 1968; 4.5 kg a.e. ha^{-1}; rooted aquatic, field: Bowmer *et al.* 1991; rate not stated; terrestrial, field)	**Poor** (Spencer 1968; 4.5 kg a.e. ha^{-1}; rooted aquatic, field)
2,4-D (PGBEE) + dichlobenil (liquid)	1	**Excellent** (Lapham 1964; 9 + 6.7 kg a.e. ha^{-1}, sequential application; floating aquatic, field@) **Fair** (Lapham 1964; 9 + 6.7 kg a.e. ha^{-1}, as mixture; floating aquatic, field@)		

Herbicide	No. of studies	Short term control ~4-16 WAT	Medium term control ~18-38 WAT	Long term control ~52 WAT
Ametryne	1	**Good** (Weldon and Blackburn 1968; 22.4 kg ha^{-1}; floating aquatic, greenhouse) **Fair** (Weldon and Blackburn 1968; 5.6 kg ha^{-1}; floating aquatic, greenhouse)		
Ametryne + 2,4,5-T (2-(2,4,5-trichloro-phenoxy) PGBEE	1	**Fair** (Weldon and Blackburn 1968; 5.6 + 1.1, 5.6 + 4.5 and 2.2 + 4.5 kg ha^{-1}; floating aquatic, greenhouse)		
Ametryne + 2,4-D amine	1	**Good** (Weldon and Blackburn 1968; 5.6 + 0.6 and 5.6 + 2.2 kg ha^{-1}, floating aquatic, greenhouse)	**Poor** (Weldon and Blackburn 1968; 6.7 + 0.28 kg ha^{-1}^, floating aquatic, field)	
Amitrole	1	**Fair** (Spencer 1968; 4.5 kg a.e. ha^{-1}; rooted aquatic, field)	**Poor** (Spencer 1968; 4.5 kg a.e. ha^{-1}; rooted aquatic, field)	**Poor** (Spencer 1968; 4.5 kg a.e. ha^{-1}; rooted aquatic, field)
Amitrole + diquat	1	**Poor** (Spencer 1968; 2.2 + 2.2 kg a.e. ha^{-1}; rooted aquatic, field)	**Poor** (Spencer 1968; 2.2 + 2.2 kg a.e. ha^{-1}; rooted aquatic, field)	**Poor** (Spencer 1968; 2.2 + 2.2 kg a.e. ha^{-1}; rooted aquatic, field)
Amitrole + picloram	1	**Fair** (Spencer 1968; 2.2 + 2.2 kg a.e. ha^{-1}; rooted aquatic, field)	**Poor** (Spencer 1968; 2.2 + 2.2 kg a.e. ha^{-1}; rooted aquatic, field)	**Poor** (Spencer 1968; 2.2 + 2.2 kg a.e. ha^{-1}; rooted aquatic, field)
Dicamba (dimethyl-amine)	2	**Excellent** (Langeland 1986a; 4.8 g a.e. L^{-1}; rooted aquatic, field)	**Poor** (Langeland 1986a; 4.8 g a.e. L^{-1}; rooted aquatic, field: Bowmer *et al.* 1991; rate not stated; terrestrial, field)	
Dichlobenil – Granular	3		**Excellent** (Weldon *et al.* 1968; 5.6 and 11.2 kg ha^{-1}; rooted aquatic, field: Bowmer *et al.* 1991; 13.5 to 20.25 kg a.i. ha^{-1}; terrestrial, field)	**Excellent** (Blackburn and Durden 1974; 4.5 kg ha^{-1}; rooted aquatic, field)
Diquat dibromide	3	**Poor** (Blackburn, 1963; 5.6 and 22.4 kg ha^{-1}; floating aquatic, greenhouse: Spencer 1968; 2.2 kg a.e. ha^{-1}; rooted aquatic, field: Kay 1999; 8.36 kg a.i. ha^{-1}; rooted aquatic, field)	**Poor** (Spencer, 1968; 2.2 kg a.e. ha^{-1}; rooted aquatic, field: Kay 1999; 8.36 kg a.i. ha^{-1}; rooted aquatic, field)	**Poor** (Spencer 1968; 2.2 kg a.e. ha^{-1}; rooted aquatic, field)
Endothal	2	**Fair** (Blackburn 1963, 22.4 kg ha^{-1}; floating aquatic, greenhouse) **Poor** (Blackburn 1963, 5.6 kg ha^{-1}; floating aquatic, greenhouse: Spencer 1968; 11.2 kg a.e. ha^{-1}; rooted aquatic, field)	**Poor** (Spencer 1968; 11.2 kg a.e. ha^{-1}; rooted aquatic, field)	**Poor** (Spencer 1968; 11.2 kg a.e. ha^{-1}; rooted aquatic, field)
Fenac	1	**Fair** (Spencer 1968; rate not specified; rooted aquatic, field)	**Poor** (Spencer 1968; rate not specified; rooted aquatic, field)	**Poor** (Spencer 1968; rate not specified; rooted aquatic, field)
Fenac + diquat	1	**Poor** (Spencer 1968; rate not specified; rooted aquatic, field)	**Poor** (Spencer 1968; rate not specified; rooted aquatic, field)	**Poor** (Spencer 1968; rate not specified; rooted aquatic, field)
Fluridone (4AS)	1		**Poor** (Langeland 1986a; 2.4 g a.i. L^{-1}; terrestrial, field)	
Glyphosate (isopropyl-amine salt) + pelargonic acid	1	**Fair** (Kay 1999; 8.1 g a.e. L^{-1} + 1.5-3%; rooted aquatic, field)	**Poor** (Kay 1999; 8.1 g a.e. L^{-1} + 1.5-3%; rooted aquatic, field)	
Glyphosate N -(phosphon-omethyl) glycine	2	**Excellent** (Langeland 1986a; 6 g a.e. L^{-1}*; floating aquatic, field: Emerine *et al.* 2010; 2.24 kg a.e. ha^{-1}; terrestrial, greenhouse)	**Poor** (Langeland 1986a, 3.6, 6 and 9.6 g a.e. L^{-1}; rooted aquatic, field)	

Herbicide	No. of studies	Short term control ~4-16 WAT	Medium term control ~18-38 WAT	Long term control ~52 WAT
Glyphosate (isopropyl amine salt)	4		**Excellent** (Sandberg and Burkhalter 1983; 3.6, 5.4 and 7.2 g a.i. L^{-1}; terrestrial, field@: Bowmer *et al.* 1991: 3.3 kg a.i. ha^{-1}; terrestrial, field) **Fair** (Sandberg and Burkhalter 1983; 3.6, 5.4 and 7.2 g a.i. L^{-1}; rooted aquatic, field@: Bowmer *et al.* 1991, 2.2 kg a.i. ha^{-1}; terrestrial, field)	**Good** (Schooler *et al.* 2008; 1.8 and 3.6 kg a.i. ha^{-1}^; terrestrial, field; below ground biomass) **Fair** (Sandberg and Burkhalter 1983; 3.6, 5.4 and 7.2 g a.i. L^{-1}; terrestrial, field@) **Poor** (Sandberg and Burkhalter 1983; 3.6, 5.4 and 7.2 g a.i. L^{-1}; rooted aquatic, field@: Schooler *et al.*, 2008; 1.8 and 3.6 kg a.i. ha^{-1}^; terrestrial, field; above ground biomass: Hofstra and Champion, 2010; 6.4 kg a.i. ha^{-1}; rooted aquatic, outdoor tanks)
Glyphosate N -(phosphon-omethyl) glycine + fluridone (4AS)	1	**Excellent** (Langeland 1986a; 3.6 g a.e. L^{-1} + 2.4 g a.i. L^{-1}; terrestrial, field)		**Poor** (Langeland 1986a; 3.6 g a.e. L^{-1} + 2.4 g a.i. L^{-1}; terrestrial, field)
Granular karbutilate	1			**Excellent** (Blackburn and Durden 1974; 33.6 kg ha^{-1}; terrestrial, field)
Imazamox	1	**Excellent** (Emerine *et al.* 2010; 560 g a.e. ha^{-1}; terrestrial, shadehouse)		
Imazapyr	5	**Excellent** (Emerine *et al.* 2010; 0.5 kg ha^{-1}; terrestrial, greenhouse: Allen *et al.* 2007; 1.2, 2.4 and 3.6 L ha^{-1}; marshes, field)	**Excellent** (Langeland 1986a; 0.6 and 1.2 g a.e. L^{-1}; rooted aquatic, field: Bowmer *et al.* 1991; 0.5 kg a.i. ha^{-1}; terrestrial, field) **Good** (Bowmer *et al.* 1991; 0.25 kg a.i. ha^{-1}; terrestrial, field)	**Excellent** (Langeland 1986a; 0.6 and 1.2 g a.e. L^{-1}*; rooted aquatic, field: Allen *et al.* 2007; 3.6 L ha^{-1}; marshes, field) **Good** (Allen *et al.* 2007; 2.4 L ha^{-1}; marshes, field: Hofstra and Champion 2010; 0.24 and 0.48 kg a.i. ha^{-1}; rooted aquatic young plants, outdoor tanks: Hofstra and Champion, 2010; 0.16 and 0.48 kg a.i. ha^{-1}*; rooted aquatic old plants, outdoor tanks) **Poor** (Hofstra and Champion 2010; 0.16 and 0.48 kg a.i. ha^{-1}; rooted aquatic old plants, outdoor tanks)
Ipazine	1	**Good** (Blackburn 1963, 22.4 kg ha^{-1}; floating aquatic, greenhouse) **Poor** (Blackburn 1963, 5.6 kg ha^{-1}; floating aquatic, greenhouse)		
Metsulfuron-methyl	5	**Good** (Langeland 1986b; 4.52 g 100 L^{-1}; rooted aquatic, field) **Fair** (Langeland 1986b; 1.13 g 100 L^{-1}; rooted aquatic, field)	**Good** (Bowmer *et al.* 1991; 29.4 g a.i. ha^{-1}; terrestrial, field)	**Excellent** (Schooler *et al.* 2008; 24 and 48 g a.i. ha^{-1}^; terrestrial, field; above and below ground biomass: Schooler *et al.* 2010; 24 g a.i. ha^{-1}^; terrestrial, field; above and below ground biomass: Hofstra and Champion 2010; 19, 36 and 72 g a.i. ha^{-1}; rooted aquatic young plants, outdoor tanks) **Good** (Hofstra and Champion 2010; 36 g a.i. ha^{-1}*; rooted aquatic old plants, outdoor tanks) **Poor** (Hofstra and Champion 2010; 36 g a.i. ha^{-1}; rooted aquatic old plants, outdoor tanks)
Norea	1	**Poor** (Blackburn 1963; 5.6 and 22.4 kg ha^{-1}; floating aquatic, greenhouse)		

Herbicide	No. of studies	Short term control ~4-16 WAT	Medium term control ~18-38 WAT	Long term control ~52 WAT
Paraquat	3	**Fair** (Blackburn 1963, 22.4 kg ha^{-1}; floating aquatic, greenhouse) **Poor** (Blackburn 1963, 5.6 kg ha^{-1}; floating aquatic, greenhouse: Lapham 1964; 2.2 kg a.e. ha^{-1}; floating aquatic, field@: Spencer 1968; 2.2 kg a.e. ha^{-1}; rooted aquatic, field)	**Poor** (Spencer 1968; 2.2 kg a.e. ha^{-1}; rooted aquatic, field)	**Poor** (Spencer 1968; 2.2 kg a.e. ha^{-1}; rooted aquatic, field)
Paraquat + dichlobenil liquid	1	**Poor** (Lapham 1964; 2.2 + 6.7 kg a.e. ha^{-1}; floating aquatic, field@)		
Picloram	2	**Fair** (Spencer 1968; 4.5 kg a.e. ha^{-1}; rooted aquatic, field) **Poor** (Lapham 1964; 0.6 kg a.e. ha^{-1}; floating aquatic, field@)	**Poor** (Spencer 1968; 4.5 kg a.e. ha^{-1}; rooted aquatic, field)	**Poor** (Spencer 1968; 4.5 kg a.e. ha^{-1}; rooted aquatic, field)
Picloram + dichlobenil liquid	1	**Excellent** (Lapham 1964; 0.6 + 6.7 kg a.e. ha^{-1}, sequential application; floating aquatic, field@) **Fair** (Lapham 1964; 0.6 + 6.7 kg a.e. ha^{-1}, as mixture; floating aquatic, field@)		
Sodium arsenite	1	**Poor** (Spencer 1968; 11.2 kg a.e. ha^{-1}; rooted aquatic, field)	**Poor** (Spencer 1968; 11.2 kg a.e. ha^{-1}; rooted aquatic, field)	**Poor** (Spencer 1968; 11.2 kg a.e. ha^{-1}; rooted aquatic, field)
Sulfo-meturon-methyl	2	**Fair** (Langeland 1986b; 0.7 g a.i. L^{-1}; rooted aquatic, field) **Poor** (Langeland 1986b; 0.3 g a.i. L^{-1}; rooted aquatic, field)	**Excellent** (Bowmer *et al.* 1991; 0.25 and 0.5 kg a.i. ha^{-1}; terrestrial, field)	
Triclopyr triethyl-amine	4	**Excellent** (Langeland 1986b; 0.36 kg 100 L^{-1}; rooted aquatic, field)	**Excellent** (Allen *et al.* 2007; 1.4, 2.8, 4.1 kg a.i. ha^{-1}; marshes, field)	**Excellent** (Schooler *et al.* 2008; 133 and 266 g a.i. ha^{-1}^; terrestrial, field; above and below ground biomass: Hofstra and Champion 2010; 13 kg a.i. ha^{-1}*; rooted aquatic young plants, outdoor tanks) **Fair** (Allen *et al.* 2007; 4.1 kg a.i. ha^{-1}*; marshes, field: Hofstra and Champion 2010; 6.5 and 9.7 kg a.i. ha^{-1}*; rooted aquatic young plants, outdoor tanks) **Poor** (Hofstra and Champion 2010; 6.5 kg a.i. ha^{-1}*; rooted aquatic old plants, outdoor tanks)
Triclopyr triethylamine + picloram	1			**Fair** (Hofstra and Champion 2010; 0.24 + 0.12 and 0.48 + 0.24 kg a.i. ha^{-1}; rooted aquatic young plants, outdoor tanks)
Various †	1		**Poor** (Bowmer *et al.* 1991; various rates†; terrestrial, field)	

Symbols and abbreviations: WAT = weeks after treatment; * = two herbicide applications (either in same year or over 2 years); ^ = more than two herbicide applications per year; # = below ground biomass reduction; @ = information on plot replication or statistical analyses not provided; a.i. = active ingredient; a.e. = acid equivalent; a.i. or a.e. only presented where provided by the cited study; **Excellent** = 90–100% control; **Good** = 80–89% control; **Fair** = 60–79% control; **Poor** = <60% control.

† Picloram + 2,4-D; triclopyr; amitrole + each of the three preceeding herbicides; hexazinone; diquat followed by glyphosate + dicamba; mowing followed by glyphosate + dicamba; four proprietary formulations of sulfonylurea herbcides (Logran® 750 g triasulfuron kg^{-1}, Harmony® 682 g thifensulfuron methyl kg^{-1} + 68 g metsulfuron methyl kg^{-1}, Express® 750 g tribenuron methyl kg^{-1}, Matrix® 500 g ethofumesate L^{-1}) + combination with 2,4-D amine; oxyfluorfen, tebuthiuron, amitrole and chlorsulfuron

tissues (Bowmer *et al*. 1993). Another species poorly controlled by glyphosate (Horsetail, *Equisetum arvense* L.) also has poor translocation of glyphosate to the roots, while susceptible species have high translocation to roots (for discussion see Bowmer and Eberbach 1993).

Another two factors that may reduce the efficacy of glyphosate on alligator weed are exudation by roots and metabolism to non-toxic metabolites. These were investigated by Eberbach and Bowmer (1995), who found that 29% of applied glyphosate was converted to CO_2 within 14 days and released through root exudation. Although they found no evidence for breakdown of glyphosate to known metabolites in below ground tissue, they could not rule out direct breakdown into unknown metabolites (and therefore unmeasurable) or very rapid subsequent breakdown of known metabolites to unknown ones.

Translocation of other herbicides in alligator weed has also been investigated. Only 'minute' proportions of metsulfuron and bensulfuron were translocated to alligator weed's below ground parts (Bowmer *et al*. 1991). Funderburk and Lawrence (1963) found downward translocation of a number of foliar applied herbicides was poor for alligator weed. There was 'little' movement of foliage applied simazine, ametryne and prometryne; 'slight' downward movement of fenac, diquat and paraquat; and 'considerable' downward movement of 2,4-D (acid) and 2,4-D butoxyethyl ester, which accumulated in the nodal portions of the stems. Despite the effective translocation of 2,4-D, it did not result in good control of the plant (55% control at 5 ppm). In contrast, Gangstad, (1978) reported more upward movement of C^{14} labelled 2,4-D than downward movement with Earle *et al*. (1951) who reported downward movement of only one to two internodes and upward movement to the top of the plants.

Zurburg *et al*. (1961) observed that phenoxy herbicides rapidly kill top parts of alligator weed but regrowth usually occurs within 5 to 8 weeks from viable apical buds. They collected stem material from treated alligator weed and found that regrowth occurred from those buds that were below the water line at the time of treatment, indicating that the water protects submersed stems from herbicide and that there was ineffectual translocation down from the aerial portions of the stems. This is somewhat in disagreement with Funderburk and Lawrence (1963), who report 'considerable' movement of 2,4-D acid and ester in alligator weed.

Observation of rapid regrowth from axillary buds on stems in herbicide-treated alligator weed (Pate *et al*. 1965, Langeland 1986a, Dugdale *et al*. 2010, Clements *et al*. 2012) indicates a further potential mode of tolerance to herbicides. Pate *et al*. (1965) found that herbicide damage by dichlobenil and dicamba did not occur in inactive axillary buds, whose vascular connections with the stem were not differentiated, apparently preventing expression of the herbicidal activity. This represents a further example where poor translocation of herbicide within alligator weed allows parts of the plant to survive herbicide application.

Rapid leaf and nodal abscission as a response to herbicide application has been observed by Dugdale *et al*. (2010) and Langeland (1986a) who speculated that much of the herbicide may be lost, or prevented from further translocation away from the parts of the plant that are directly exposed to the herbicide spray (i.e. under the canopy, the water or the ground), by auto-abscission of the parts of these plant tissues that have the highest herbicide concentrations. Therefore, active abscission of exposed tissues may be an additional factor that provides alligator weed with herbicide tolerance. An alternative explanation may be that, because herbicide activity is quite commonly concentrated at nodes, the herbicide damages vascular tissues here and prevents further translocation and results in stems breaking at the nodes.

In summary, there are a number of factors that probably result in the tolerance of alligator weed to herbicides: 1) poor translocation out of leaves; 2) poor translocation to roots resulting in sublethal tissue concentrations; 3) exudation of glyphosate (and possibly other herbicides) by underground tissues (possibly with intermediary metabolites); 4) poor translocation to quiescent buds; and 5) auto-abscission or breakage of nodal tissue with high herbicide concentrations.

All of the above translocation studies have been undertaken on foliar applied herbicides. Interestingly, Solymosy (1968) has shown that absorption and translocation of herbicide through alligator weed roots is very efficient and in some cases may be much better than foliar absorption. Such a method provides a potentially great benefit, by simultaneously circumventing the problem of poor translocation and the problem of poor herbicide coverage on underwater plant parts. Granular dichlobenil application does fit this mode of application and provides excellent control of rooted-emerged alligator weed but not mats of floating alligator weed (Weldon *et al*. 1968). Additional research on efficacy of herbicides taken up by roots would be valuable.

Control programs

This section summarises available literature on control programs for alligator weed from around the world. The aim is to give an indication of what has been done and with what degree of success to guide weed managers in making decisions about alligator weed control. By virtue of the authors' affiliations, more information is provided on Australian and New Zealand control programs than those from other countries. Such an approach does not allow for direct comparisons between programs or herbicides used.

United States of America

Alligator weed was first recognised as a problem at the start of the 20th century in Alabama, where it was recorded completely filling a creek in 1897 (Durden *et al*. 1975) and in 1901 Major William Rossel, United States Army Corps of Engineers, recognised its potential as a major obstruction to navigation and drainage. At the time, water hyacinth (*Eichhornia crassipes* (Mart.) Solms) was a superior competitor because it began its growth earlier in the growing season and so shaded out and retarded the alligator weed. However, after the systematic removal of water hyacinth with 2,4-D, alligator weed became the threat that Major Rossel had warned of (Lawrence and Funderburk 1965, Gangstad 1978). Preliminary studies were made on chemical control of water hyacinth and alligator weed in 1950 and until 1975 phenoxy herbicides were widely used to control these two species. However, herbicide application for alligator weed was quite limited because the application rate required was high (and so costly) and control was never satisfactory, usually requiring retreatment after a short period (Gangstad *et al*. 1975), although frequent applications were considered successful at reducing the spread of alligator weed (Blackburn 1963). For example, single applications of 2,4-D amine salt (9 kg ha^{-1}) gave inadequate control except where it was growing in deep water, even then requiring two applications to bring it under control (Eggler 1953). By 1975 control programs integrating herbicide (2,4-D or 2,4,5-T) with biological control (using flea beetles (*Agasicles hygrophila* Selman and Vogt), thrips (*Amynothrips andersoni* O'Neill) and moths (*Vogtia malloi* Pastrana)) had been established (Durden *et al*. 1975). Fortunately, good control of floating alligator weed was achieved with the flea beetle, particularly when integrated with herbicide programs (Durden *et al*. 1975). Attempts to establish the alligator weed flea beetle have been unsuccessful in North Carolina due to cold sensitivity (Langeland 1986a). This means that floating aquatic alligator weed poses a much greater problem in these areas and herbicide control programs are required for the aquatic ecotype.

In the 1960's repeat applications of picloram and picloram + 2,4-D were used in Louisiana (Lapham 1966). Then, 2,4,5-T (PGBEE) was considered the only consistently effective herbicide for control of alligator weed. Approximately 27,000 kg active ingredient (a.i.) were used in 1978 to treat ~4,000 ha of alligator weed. However, because of concerns over its environmental and health risks, all uses of it were banned by the United States Environmental Protection Agency in 1983 (Gangstad 1984).

Langeland (1986a) reviewed the alligator weed control program in North Carolina. He reports 2,4,5-T (PGBEE) has been effective in controlling alligator weed in many areas but not in North Carolina. Based on an assessment of 3,000 herbicide treatments that were evaluated over a five year period in North and South Carolina; 2,4,5-T was the most consistent of these, but still provided less than 50% control when applied as a spray solution. Granular application of 2,4,5-T gave 'acceptable' control when alligator weed was emersed, flowering and rooted. 'Excellent' control was achieved with 2,4-D and fenac applied in combination but only when applied 8-10 weeks before frost and then flooded just after treatment. Other herbicides to be evaluated in the program were: 2,4,5-T; 2,4-D; MCPA; [2(2,4-DP); dalapon]; silvex esters and amides; fenac; amitrole and 2,3,6-TBA; TCA; AMS; endothall; simazine; neburon; fenuron; monuron; diuron; monuron TCA; fenuron TCA and erbon. The results were erratic with effective control only occurring when flooding occurred shortly after herbicide application. Control was 90 to 100% for maleic hydrazide, amitrole, dalapon, erbon, silvex, MCPA and 2,4-D acetamide but highly ineffective in areas not subject to flooding (Langeland 1986a). This suggests that the stress of being flooded acts in combination with the stress provided by the herbicide. Stresses likely to be caused by flooding include anoxia, reduced light and therefore photosynthetic capacity, and reduced and altered gas exchange.

Later herbicide evaluations in North Carolina indicated that glyphosate could be used in management programs to control alligator weed growing in lakes and rivers, while imazapyr was most suitable for non-irrigation ditch banks (Langeland 1986a). Preliminary investigations also found that triclopyr was very effective and could be useful for aquatic sites and ditch banks (Langeland 1986a). Langeland (1986a) recommended two applications of glyphosate in the first year of treatment, at recommended rates, in lakes and rivers. For ditch banks, he recommended imazapyr at 0.06 to 0.12 g a.e. (acid equivalent) 100 L^{-1} be applied on a spray to wet basis. He suggested that with two consecutive applications, alligator weed will be "essentially eliminated" from some ditch banks with re-colonisation occurring from dispersal of vegetative propagules. After this initial effort an annual visit, at most, to spot spray would be required. Triclopyr amine and imazapyr are currently used in control programs to suppress alligator weed (Allen *et al*. 2007).

New Zealand

In New Zealand, alligator weed control is undertaken for suppression or eradication, depending on location, and most control is carried out with herbicides. Metsulfuron-methyl is used at aquatic and urban sites, glyphosate is used for the aquatic sites (often in conjunction with metsulfuron-methyl) and triclopyr ethoxyethyl ester with picloram at pasture and cropping sites. The alligator weed management programs are reducing both the number and extent of alligator weed sites, although it is still regarded as a problematic plant by waterway managers with limited or ineffective control options (Hofstra and Champion 2010). There are no products that provide effective reduction of alligator weed biomass over a season with one application (Hofstra and Champion 2010) so the control programs require two or three herbicide applications per year to be effective (Champion 2008). A key to the success of this program is that site-specific management plans are developed, with control methods differing depending on whether sites are aquatic or terrestrial, urban or rural, cropping or pasture, risk of spread and potential vectors. Other key drivers of success are the experience of the personnel contributing to the program, sufficient funding, relevant permits to allow appropriate herbicides to be used, and use of legislation to prevent movement of contaminated soil, machinery or garden waste (Champion 2008). Aquatic sites are treated with aerial, boat or land-based hose applications of metsulfuron-methyl or glyphosate. Urban sites are treated by injecting metsulfuron-methyl into the hollow stems to provide control without damaging surrounding vegetation. The propriety mixture of picloram and triclopyr ethoxyethyl ester gives the best results in terrestrial situations, with significant reductions in abundance at all sites treated. Wherever an intensive program of treatment and follow-up has been implemented, the near eradication of alligator weed has been achieved over a two to three year period. Careful manual excavation of remaining tap roots at terrestrial sites is recommended when sites are reduced to the last few remaining plants (Champion 2008).

Australia

In Australia, control of alligator weed with herbicide is the most commonly used method, while physical and mechanical removal are also widely adopted (Burgin *et al*. 2010). Despite widespread and intensive control in the state of New South Wales, there was an increase in the number and extent of infestations between 2001 and 2007 (Burgin *et al*. 2010). Control of aquatic alligator weed is achieved with the alligator weed flea beetle in large infestations with permanent water in warmer areas of Australia but it fails in temperate areas (i.e. Victoria and southern New South Wales), in small or ephemeral waterways and in terrestrial situations (van Oosterhout 2007). In some situations, where alligator weed is present in a small part of its potential range, control programs may aim to eradicate all alligator weed from a region or jurisdiction. This represents a much more difficult target than maintaining biomass at low levels.

In efforts to improve alligator weed control, multiple herbicide applications per year are used (van Oosterhout 2007) in a resource depletion strategy. A comprehensive management strategy is in place in Australia where infestations are regarded as 'core' or 'non-core' (van Oosterhout 2007). The former are areas where eradication is not feasible and they are managed to reduce abundance and prevent further spread. Aquatic infestations in the warmer climate 'core' areas are suppressed by the flea beetle. For 'non-core' areas, suppression leading to eradication or immediate eradication are the goals. Manual excavation is recommended for small patches, while larger patches should be sprayed with a broad spectrum herbicide (to allow easy detection of alligator weed regrowth) and retreated at least three times per year. Complete eradication of these sites has proven difficult so excavation of the remnant plants is suggested once the infestation becomes small. In terrestrial areas, selective herbicides are recommended to encourage competition from monocots.

The eradication approach in Australia acknowledges that translocation of herbicide to the underground portions of the plant is poor and regrowth will occur. The eradication approach is to damage above ground stems by repeated and frequent herbicide application with three applications of metsulfuron-methyl per year, typically for six years. After each herbicide application it is expected that the plant will respond by establishing new shoot growth, fuelled by energy and nutrient reserves in the underground roots and rhizomes. Because the reserves are depleted regrowth may be slow after two years, and from then on only two applications per year may be possible. If each successive application is applied before there is significant downward translocation of carbohydrates and nutrients, then the plant will eventually exhaust its reserves and die (van Oosterhout 2007). However, if the frequency of herbicide application is such that it always or occasionally allows replenishment of the underground parts, eradication will not be achieved. Although we are unable to say in general terms how many applications are required over what period to achieve sufficient depletion to kill an infestation, nor how this might vary in different climatic zones, evidence of nutrient and biomass depletion in roots has been obtained by Schooler *et al*. (2007, 2008, 2010).

Schooler *et al*. (2008) also provides evidence for the resource depletion strategy where suppression is the primary target in certain 'non-core' areas. Here the regular application of dicotyledon-specific herbicides is recommended. Schooler *et al*. have shown in a temperate pasture situation that dicotyledon-specific herbicides (metsulfuron-methyl and triclopyr triethylamine) resulted in a greater reduction in alligator weed biomass over the long term (15 months after final treatment) and an increase in monocot biomass compared to the broad spectrum herbicide used (glyphosate). This was attributed to competition between the grasses and the alligator weed and suggests that prevention of spread may be enhanced when using dicotyledon-specific herbicides. Further, in terms of suppressing alligator weed, they found that there was no advantage of four applications over three, nor was there an advantage of using high rates over low, so they recommended applying low concentrations of selective herbicides relatively infrequently (one to three times per year).

Cook and van Oosterhout (2008) provide a technique to suppress alligator weed in pasture based on tipping the competitive advantage from alligator weed to pasture species. It is achieved by reducing the canopy height by slashing or grazing, thus promoting new growth of alligator weed and more importantly opening the pasture canopy to allow greater herbicide coverage on the alligator weed; metsulfuron-methyl is then applied 2-4 weeks later, which kills much of the alligator weed and allows the pasture species to increase in abundance. This process is repeated three times per year and can maintain alligator weed cover <10%. A prerequisite for success is that there must be at least 10% cover of a stoloniferous grass that grows to at least 30 cm tall (i.e. kikuyu (*Pennisetum clandestinum* Hochst. ex Chiov.), Swazi grass (an improved variety of *Digitaria didactyla* Willd., formerly *D. swazilandensis* Stent) and Rhodes grass (*Chloris gayana* Knuth.), to provide strong competion to the alligator weed.

Based on success in the USA (e.g. Allen *et al*. 2007), a permit has recently been granted to use imazapyr in New South Wales to control alligator weed in terrestrial situations, but it is too early to determine how successful this has been. Glyphosate and dichlobenil have also been used for eradication and suppression programs in Australia, but these are not recommended for eradication programs. Glyphosate has not been as effective as metsulfuron-methyl, and dichlobenil suppresses alligator weed growth near the soil surface, but when the effect of this residual herbicide ceases, regrowth occurs from material below the strata that the herbicide penetrates to. A mix of glyphosate and metsulfuron has also been used in the past to little effect (van Oosterhout 2007).

In Victoria, initial treatment programs in urban backyards usually used dichlobenil (4.05 kg a.i. ha^{-1}), but metsulfuron-methyl (0.06 g a.i. L^{-1}), glyphosate (3.6 g a.i. L^{-1}), or a mixture of the later two herbicides were also used (Gunasekera and Adair 1999). The treatment program began in 1997/1998 and 65 to 75% of 800 infestations were successfully "eradicated" by a single treatment with dichlobenil (eradication was defined as the weed not being present on a subsequent visit, Gunasekera *et al*. 2006). By 2008, only five backyard sites out of a total of 805 were thought to contain regrowth following initial treatment in 2006/2007 (Gunasekera and Bonilla 2008). However, a high proportion (>50%) of these sites were found to still have alligator weed in subsequent site visits during 2009/2010 (E. Cox, Department of Primary Industries Victoria, personal communication). Despite being implicated in eradciation of ~50% of Victroian sites, dichlobenil is currently not recommended as a tool for the eradication of alligator weed because it is a residual herbicide, suppressing the subterranean roots for some time, rather than killing it, and resulting in the site being declared eradicated (van Oosterhout 2007). However, its residual nature may be an advantage, because it needs to be applied less frequently, and all of the few efficacy studies in the literature report excellent medium and long term control (Table 1). Therefore, with proper monitoring programs this effective herbicide need not be excluded from use.

Control of alligator weed infestations in urban backyards has been undertaken in the Hawkesbury-Nepean Catchment, New South Wales. In 2004, re-inspections of 66 of the backyards where alligator weed has been controlled in the past revealed 30 infestations that were still present (45%). Metsulfuron-methyl was used to control alligator weed in 80% of these cases, with glyphosate and a combination of MCPA and dicamba also used. All three herbicides were effective in controlling the above ground parts of the plant in the short term but no information is provided regarding long term control (Meyer *et al*. 2007).

For aquatic sites, aquatic formulation glyphosate is a good candidate for alligator weed control. The reasons for this are that it: is effective at killing other perennial weeds with underground storage organs; is generally translocated within a few hours from the foliage to the roots, has low aquatic toxicity; and rapidly deactivates in the soil (Bowmer *et al*. 1993). Indeed, it has proven effective at aquatic sites in the USA (Langeland 1986a) and at controlling floating mats of alligator weed in Australia. Free floating alligator weed was controlled effectively using standard rates of glyphosate (3.6 g a.i. L^{-1}) in Botany Wetlands in Sydney and at Barren Box swamp in Griffith (both New South Wales; Sainty *et al*. 1998). Glyphosate is far less effective on terrestrial or rooted alligator weed, which have an extensive underground root system making them harder to control (Cook and Storrie 2008, Anonymous 1994). Bowmer *et al*. (1993) used glyphosate at high volume (400 L ha^{-1} or up to 3.2 kg ha^{-1}) in late spring, late summer or autumn. Excellent short term control was achieved, but regrowth always occurred.

At a high value site in Sydney that consisted of 11 interconnected ponds and dams, spot-treatments of herbicide were applied at regular intervals for ten years. On average, application occurred at monthly intervals during cooler months and bi-monthly during warmer months (September to May). Aquatic infestations have been treated with 3.6 g a.i. L^{-1} glyphosate while terrestrial infestations have been treated with metsulfuron-methyl (rate not specified). Where possible, water levels were lowered to increase the amount of alligator weed exposed to herbicides. In addition, manual removal of floating mats occurred. Over ten years this has resulted in an estimated 90 to 95% control of the initial alligator weed infestation (from 5,000 m^2 to less than 500 m^2, Chandrasena and Pinto 2007). The authors suggested that poor control was achieved with glyphosate because of inadequate plant uptake. Often there were few leaves exposed above the water to apply herbicide to, access was difficult in soft mud and poor retention of spray on the leaves was observed. Such sub-lethal doses caused stem fragmentation that increased spread. Apparently, much better control was observed with metsulfuron-methyl. The description of the alligator weed at this site indicates that the remaining alligator weed was rooted into the substrate and therefore it was expected to be much more difficult to control than the floating mats described by Sainty *et al*. (1998).

In Victoria, the application of glyphosate (3.6 g a.i. L^{-1}) at two-monthly intervals during the growing season over several years was generally needed to eradicate an average sized infestation (10 m^2) growing along the margins of watercourses (Gunasekera *et al*. 2006; for these naturalised infestations, unlike the previously mentioned program from this paper, "eradication" was considered achieved when two repeat visits failed to find alligator weed). However, many sites in Victoria remain intact, though smaller in size. A recent review of the program has shown that in all cases regular applications of glyphosate or metsulfuron-methyl resulted in substantial reductions in abundance and cover but that eradication was not achieved (Dugdale *et al*. 2008). Like Sainty *et al*. (1998), Dugdale *et al*. (2008) found the problem lay in finding and killing the last remaining plants (which may be dormant roots or shooting plants hidden by competing vegetation).

Although glyphosate and metsulfuron together provide good control of alligator weed in aquatic and terrestrial situations, eradication of infestations remains problematic. Julien and Bourne (1988) report that six of 33 sites were eradicated, however the importance of herbicide in achieving this is probably minor. Four of these eradications were achieved by excavation, one by a combination of residual herbicide and excavation, and one by a single application of herbicide (dicamba). The later site was a creek bed site of 50 m^2 and scouring of the creek bed was suspected in contributing to its eradication. It should be noted that the information provided in Julien and Bourne (1988) is not detailed enough to know if the herbicides were used appropriately, i.e. applied repeatedly over several years in a resource depletion strategy. Therefore, we do not know if eradcaition failed at the 28 other sites that were treated with herbicide because the herbicides (the actual herbicides used were not listed) were ineffective or because they were not applied properly. As already stated, a major problem with achieving eradication is reliably finding and killing the last plants.

A number of similar case histories were summarised by Sainty *et al*. (1998). For example, at Woomargama, New South Wales, glyphosate was applied repeatedly for 20 years but shoots continued to sprout (the frequency of application was not provided by Sainty *et al*. 1998). This site remains under a control program. Other herbicides applied there included 2,4,5-T, flupropanate and tetrapion (Julien and Bourne 1988). At Barren Box swamp near Griffith, many hectares of free-floating alligator weed were controlled using a single application of glyphosate in the mid 1990's. Rooted alligator weed growing along the 22 km perimeter of the swamp required three herbicide applications per year for three growing seasons. In the first two years, a tank mix of glyphosate and metsulfuron at recommended rates was used to kill *Typha* spp. that the alligator weed was growing amongst. In the third year, metsulfuron was used alone. Alligator weed has not yet been eradicated from this site.

In response to poor control in Australia, Chandrasena *et al*. (2004) suggested that site specific management plans need to be developed for each site to achieve better control. This was enacted in New Zealand, and where they exist, excellent control of alligator weed was achieved (Champion 2008).

China

Although significant resources are directed to alligator weed management in China (US$72 million per annum; Liu and Diamond 2005) there are no publications that describe control programs.

Summary

The control of alligator weed with herbicide is challenging. Alligator weed has an extensive underground root system and one of the key impediments to successful control is poor translocation of foliar applied herbicides to below ground portions of the plant. There are a number of factors that probably result in the tolerance of alligator weed to herbicides, these being: 1) poor translocation out of leaves; 2) poor translocation to roots resulting in sub-lethal tissue concentrations; 3) exudation of glyphosate (and possibly other herbicides) by underground tissues (possibly with intermediary metabolites); 4) poor translocation to quiescent buds; and 5) auto-abscission or breakage of nodal tissue with high herbicide concentrations. There is some evidence that soil-applied herbicide provides better control than foiliar-applied herbicide, because the issue of poor translocation is circumvented. However, additional research is required to confirm this.

The efficacy of forty-nine herbicides, or herbicide combinations, have been reported in the literature, but many more have been tested without rigour or sufficient clarity to be included here. Floating aquatic alligator weed can be controlled effectively with glyphosate (Langeland 1986a, Sainty *et al.* 1998, Chandrasena and Pinto 2007), although management of subsequent fragments may be needed (Dugdale *et al.* 2010, Clements *et al.* 2012). However, almost universally, control of rooted alligator weed has been difficult to achieve, with most herbicides applied once or infrequently only scorching the above ground or above water parts of the plant. Regeneration proceeds rapidly, typically from roots and rhizomes that are protected from herbicide application (by the soil or water shielding them and poor translocation through the plant). Substantial biomass reductions are achieved when multiple applications of herbicide are made per year with the best herbicides being imazapyr and metsulfuron-methyl, although satisfactory results have also been reported with glyphosate, and triclopyr TEA. Despite the wealth of information available, it is difficult to determine which herbicide is the best because, the impacts of herbicides on key alligator weed response metrics (above ground biomass, below ground biomass and stem and root viability) are unknown for many herbicides. For example, it is not known if viable stem fragments are produced after application for herbicides other than glyphosate and metsulfuron-methyl, and the effect of single or multiple herbicide applications on below ground biomass and root viability is not well documented for most herbicides. Furthermore, how these differ for aquatic versus terrestrial alligator weed is often not well understood.

Control programs rely on multiple applications. Current management approaches are based on a resource depletion strategy. Repeat herbicide applications are made at short time intervals to kill re-sprouting alligator weed before it has begun replenishing its underground reserves, thus eventually exhausting the plant. Although there is limited information on the reduction of below ground biomass following herbicide application, the studies of Schooler *et al*. (2008) and Bowmer *et al*. (1991) provide strong evidence that the resource depletion approach to eradicating terrestrial alligator weed is effective at reducing below ground biomass and viability. Despite this, eradication remains difficult to achieve with herbicides alone and mechanical removal of the last remaining plants is sometimes required.

Acknowledgements

We thank Melbourne Water and the Department of Primary Industries Victoria for funding this project and Trevor Hunt and Daniel Clements for reviewing earlier drafts of the manuscript. Two anonymous reviewers provided valuable suggestions that improved this paper.

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