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**Field Release of
Ceratapion basicorne
(*Coleoptera: Apionidae*), a
Weevil for Biological
Control of Yellow
Starthistle (*Centaurea
solstitialis*), in the
Continental United States**

**Environmental Assessment,
August 2019**

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basicorne* (Coleoptera:
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in the Continental United States**

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I. Purpose and Need for the Proposed Action

The U.S. Department of Agriculture (USDA), Animal and Plant Health Inspection Service (APHIS), Plant Protection and Quarantine (PPQ), Pests, Pathogens, and Biocontrol Permits (PPBP) is proposing to issue permits for release of a weevil, *Ceratapion basicorne* (Illiger) (Coleoptera: Apionidae), in the continental United States. This agent would be used for the biological control of yellow starthistle, *Centaurea solstitialis* L. (Asteraceae). Before permits are issued for release of *C. basicorne*, APHIS must analyze the potential impacts of the release of this organism into the continental United States.

Classical biological control of weeds is a weed control method where natural enemies from a foreign country are used to reduce exotic weeds that have become established in the United States. Several different kinds of organisms have been used as biological control agents of weeds: insects, mites, nematodes, and plant pathogens. Efforts to study and release an organism for classical biological control of weeds consist of the following steps (TAG, 2016):

1. Foreign exploration for potential biological control organisms in the target weed's area of origin.
2. Host specificity studies to ensure that the organism is specific to the target weed and will have minimal impact on non-target plants.
3. Approval of the organism (permit issuance) by the PPBP for release into the environment of the United States.
4. Release of the organism by the permittee and establishment of the organism in areas of the United States invaded by the target weed.
5. Post-release monitoring of the impacts of the biological control organism (conducted by the permittee).

This environmental assessment¹ (EA) has been prepared, consistent with USDA, APHIS' National Environmental Policy Act (NEPA) implementing procedures (Title 7 of the Code of Federal Regulations (CFR), part 372). It examines the potential effects on the quality of the human environment that may be associated with the release of *C.*

¹ Regulations implementing the National Environmental Policy Act of 1969 (42 United States Code 4321 *et seq.*) provide that an environmental assessment "[shall include brief discussions of the need for the proposal, of alternatives as required by section 102(2)(E), of the environmental impacts of the proposed action and alternatives, and a listing of agencies and persons consulted." 40 CFR § 1508.9.

basicorne to control infestations of yellow starthistle within the continental United States. This EA considers the potential effects of the proposed action and its alternatives, including no action.

The applicant's purpose for releasing *C. basicorne* is to reduce the severity and extent of yellow starthistle in the continental United States. Yellow starthistle, *Centaurea solstitialis* L., originates from the Mediterranean region, where it generally occurs in low densities and appears to be under natural control (Uygun et al., 2004). It was accidentally introduced into California over 130 years ago, primarily through importation of contaminated alfalfa seed (Maddox et al., 1985). The weed infests about 8 million hectares (16–20 million acres) in the western United States and Canada (Duncan, 2001; Pitcairn et al., 2006). It has been reported in 41 of the 48 contiguous U.S. states, with the heaviest infestations in the states of California, Idaho, Oregon, and Washington (Maddox et al., 1985). It is the most common weed in California, and it is continuing to spread (Pitcairn et al., 2006). Yellow starthistle is highly invasive in grassland habitats and displaces desirable plants in both natural and grazing areas. Its flowers have inch-long spines that deter feeding by grazing animals and lower the utility of recreational lands. Consumption of yellow starthistle by horses causes a fatal syndrome known as "chewing disease" (Cordy, 1978). Conventional control strategies have been inadequate because of the size of the infestation, economic and environmental costs of herbicides, and the relatively low monetary return from grazing and recreational land use.

There is a need to release a host-specific biological control agent to reduce infestations of yellow starthistle because current controls have not been effective against this widespread weed. The objective of releasing *C. basicorne* is to decrease the size and number of yellow starthistle plants. The weevil does not directly kill the plant, but stunts the growth of the plant, reducing development of flowers and seeds (which decreases the subsequent generation). The weevil is not expected to cause extinction of yellow starthistle, but it is expected to continue impacting the weed year after year.

Public involvement

Notice of this EA was made available in the Federal Register on March 11, 2009 for a 30-day public comment period. One comment was received on the EA from the Tribal Historic Preservation Officer of the Bois Forte Band of Ojibwe in Minnesota. The commenter indicated a general disagreement with biological control but did not raise any specific issues regarding the release of the organism.

II. Alternatives

This section will explain the two alternatives available to the PPBP; no action and to issue permits for release of *C. basicorne*. Although the PPBP's alternatives are limited to a decision on whether to issue permits for release of *C. basicorne*, other methods available for control of yellow starthistle are also described. These control methods are not decisions to be made by the PPBP and may continue whether or not permits are issued for environmental release of *C. basicorne*. These are methods presently being used to control yellow starthistle by public and private concerns.

A third alternative was considered, but will not be discussed further. Under this third alternative, the PPBP would have issued permits for the field release of *C. basicorne* but permits would contain special provisions or requirements concerning release procedures or mitigating measures. No issues have been raised that would indicate that special provisions or requirements are necessary.

A. No Action

Under the no action alternative, the PPBP would not issue permits for the field release of *C. basicorne* for the control of yellow starthistle. The release of this biological control agent would not take place. The following methods are presently being used to control yellow starthistle. These control methods will continue under the "No Action" alternative and may continue even if permits are issued for release of *C. basicorne*.

1. Chemical Control

A variety of herbicides can effectively kill yellow starthistle plants; however, because of expense and environmental concerns, herbicides are used primarily in highly productive farmland, on roadsides, and around the perimeter of large infestations to contain their spread (Sheley et al., 1999). Because yellow starthistle seeds can remain dormant in the soil for several years, the weed can reappear after treatment (Joley et al., 2003).

Clopyralid is the most common herbicide used in California to control yellow starthistle (DiTomaso, 2005). It has excellent soil (preemergence) and foliar (postemergence) activity, has a broad timing window, does not appear to negatively impact insect biological control agents, and has no grazing restrictions. It is slow acting and often requires two months to control starthistle and it is only effective the year that it is applied. Clopyralid is a very selective herbicide and does not injure grasses or most broadleaf species. However, depending on the timing of application, it

does damage or kill most species in the legume family (Fabaceae), as well as the sunflower family (Asteraceae), and this may not be a desired outcome in a control program with the goal of increasing native plant diversity or enhancing a threatened native plant population susceptible to the herbicide. It can also cause some injury in members of the nightshade (Solanaceae), knotweed (Polygonaceae), carrot (Apiaceae), and violet (Violaceae) families. In contrast, many other broadleaf species, including species in the mustard (Brassicaceae) and poppy (Papaveraceae) families and filarees (*Erodium* spp.) are very tolerant to the herbicide.

Picloram is the most widely used herbicide to control yellow starthistle in western states other than California, where it is not registered (DiTomaso, 2005). It acts much like clopyralid, but has a broader spectrum of control and a much longer soil residual activity. The most effective time to apply picloram is when plants are young, during late winter to early spring (Callihan et al., 1989). Treatment can provide effective control for about two to three years. Although well-developed grasses are not usually injured by proper doses, young grass seedlings with less than four leaves may be killed (Sheley et al., 1999).

2. Mechanical Control

Mechanical control methods include hand pulling, hoeing, tillage, and mowing. Manual removal of yellow starthistle is most effective with small patches or in maintenance programs where few plants occur sporadically. Tillage, using plows or discs that bury plant parts, or harrows, knives, and sweeps that damage root systems or to separate shoots from roots of younger plants, can be effective but are not appropriate for rangeland (Thomsen et al., 1996). Mowing is a popular control technique along highways and in recreational areas.

3. Cultural Control

Prescribed burning has sometimes successfully helped control yellow starthistle (Hastings and DiTomaso, 1996; DiTomaso et al., 1999). The best time for burning is usually in early to mid-summer (June to early July), after yellow starthistle has bolted but before it starts producing seeds, and when seeds of most desirable species have already dispersed and grasses have dried to provide adequate fuel. Success depends on having sufficient dry plant material to carry the fire. Because yellow starthistle is still green at this season, it is killed by scorching rather than burning, and it does not fuel the fire.

Livestock grazing can effectively reduce yellow starthistle under highly controlled conditions. Essentially, sheep, goats, and/or cattle can be used

as living mowing machines to reduce yellow starthistle foliage. Grazing pressure needs to be critically timed and managed to maximize damage to the weed while sparing the more desirable vegetation. Repeated grazing at two-week intervals has been shown to suppress growth, but it did not eliminate yellow starthistle (Thomsen et al., 1996). Intensive grazing in May and June, when yellow starthistle bolts but before it is spiny, reduced growth, canopy cover, survivability, and reproductive capacity of yellow starthistle (Thomsen et al., 1993).

Revegetation programs for yellow starthistle control are still experimental, expensive, and require long time periods. Usually they employ planting native or introduced perennial grasses by seed drill, often integrated with other control methods such as herbicides (e.g., Callihan et al., 1986; Larson and McInnis, 1989; Northam and Callihan, 1988; Prather and Callihan, 1991). Planting forage legumes in pastures has also been used with some success (Sheley et al., 1993; Thomsen et al., 1997).

4. Biological Control

A biological control program for yellow starthistle began in the 1960s (Turner et al., 1995; Piper, 2001; Pitcairn et al., 2004). Five insects, *Bangasternus orientalis* (Coleoptera: Curculionidae), *Eustenopus villosus* (Coleoptera: Curculionidae), *Larinus curtus* (Coleoptera: Curculionidae), *Urophora sirunaseva* (Diptera: Tephritidae), and *Chaetorellia australis* (Diptera: Tephritidae), have been approved for release and subsequently became established in the western United States for control of this weed. The fly *Urophora jaculata* was approved for release but failed to establish because it was not adapted to the North American population of yellow starthistle (Clement, 1994). Another fly *Chaetorellia succinea* was accidentally introduced in 1991 and has become well established (Balciunas and Villegas, 1999).

All of the introduced insects attack the flowerheads and reduce seed production (Pitcairn et al., 2002). Two insects, *Ch. succinea*, and *E. villosus* are now widespread and abundant in California and Oregon, and are increasing in Idaho and Washington (Pitcairn et al., 2003). *Bangasternus orientalis* and *U. sirunaseva* are widespread in California, but not very abundant (up to 25 percent of flower heads infested). *Larinus curtus* and *Ch. australis* are neither widely established nor abundant in California, although they are more successful in Oregon. Some populations of *L. curtus* were found infested with the internal microsporidian pathogen *Nosema* sp., which may severely reduce the weevil's reproduction. However, populations not infested with *Nosema* have also failed to achieve high numbers. *Chaetorellia australis* emerges early in the spring, long before yellow starthistle flowerheads are available

for egg laying. The first generation can reproduce in another exotic plant, *Centaurea cyanus* (bachelor's button), when it is available (Balciunas and Villegas, 1999); however, in California many flies probably die before yellow starthistle flowerheads become available. This fly has become well established only in areas infested with both yellow starthistle and bachelor's button or in some areas of Oregon where the insect and yellow starthistle are better synchronized.

The exotic rust pathogen, *Puccinia jaceae* var. *solstitialis* was approved for release in California in 2003 and was released in 20 counties in 2004 and 38 counties in 2005 (Woods and Villegas, 2004; Woods et al., 2004a). Release sites were surveyed to determine the status of the rust, and 3 years following inoculation, only 3 percent of the locations showed infection (CDFA, 2007). This suggests that the rust may be limited by environmental factors and may not establish in all areas infested with yellow starthistle (CDFA, 2007).

B. Issue Permits for Environmental Release of *Ceratapion basicorne*

Under this alternative, the PPBP would issue permits for the field release of *C. basicorne* for the control of yellow starthistle. These permits would contain no special provisions or requirements concerning release procedures or mitigating measures.

1. Biological control agent information

Taxonomy

Ceratapion basicorne (Illiger) was described in 1807 as *Apion basicorne* (Alonso-Zarazaga, 1990a). Other synonyms are *A. subdentirostre* Desbrochers, *A. simillimum* Desbrochers, *A. caullei* var. *subcavifrons* Desbrochers, *A. spathula* Desbrochers, *A. atripenne* Desbrochers, *A. tauricum* Desbrochers, *A. alliariae* Herbst, *A. caullei* var. *subcaviceps* Desbrochers, *A. spathifer* Desbrochers, *A. distans* auct. nec Desbrochers, *A. brevicorne* Megerle, and *A. intermedium* Rey. The taxonomic position of the species is (Alonso-Zarazaga, 1990b):

Phylum: Arthropoda
Class: Hexapoda
Subclass: Pterygota
Order: Coleoptera
Suborder: Polyphaga
Superfamily: Curculionoidea
Family: Apionidae
Tribe: Ceratapini
Genus: *Ceratapion*
Subgenus: *Echinostroma*
Species: *basicorne*

The genus *Ceratapion*, includes 55 species and subspecies and is the most diverse and speciose genus of the tribe Ceratapini (Alonso-Zarazaga, 1990b; Wanat, 1994). The geographical distribution of the genus covers almost the entire Palaearctic region (biogeographic region that includes Europe, the northwest coast of Africa, and Asia north of the Himalaya Mountains). The center of origin of the Ceratapini is probably in western and central Asia, coinciding with that of their Asteraceae host plants.

Geographical Range

Ceratapion basicorne is distributed throughout Europe and southwestern Asia, from Spain to Azerbaijan, between 37° and 55° N latitude (Alonso-Zarazaga, 1990a; Wanat, 1994). Specimens have been collected in: Spain, France, Belgium, Italy, Switzerland, Germany, Denmark, Sweden, Poland, Ukraine, Czech Rep., Slovakia, Austria, Hungary, Rumania, Croatia, Bosnia, Herzegovina, Albania, Bulgaria, Greece, Cyprus, Turkey, Iran, Lebanon, Israel, Azerbaijan, Nakhichevan Rep., Armenia, Georgia, Russia, and possibly Morocco and Algeria. This distribution overlaps that of yellow starthistle, including the casual populations in central and northern Europe. The insect appears to have a wide tolerance to climate, and it occurs in some regions that have cold winters with snow cover. It commonly infests yellow starthistle in Turkey and Greece (Rosenthal et al., 1994). If *C. basicorne* is released in North America, it is expected to become established throughout the range of yellow starthistle.

Life History

Ceratapion basicorne adults emerge from hibernation in the early spring and feed on yellow starthistle leaves (Clement et al., 1989). Females lay eggs in the leaves of rosettes from late March to early May in central Italy. Eggs hatch in about 10 days at room temperature, and first instar larvae mine in the leaf blade and down the petiole. Larvae feed primarily in the

root crown (upper part of root stem), complete development in about two months, and pupate inside the plant. Adults emerge in June, feed on yellow starthistle leaves for a few days then disappear. They are thought to aestivate (become dormant during the summer or dry season) and hibernate (overwinter in a dormant state) in secluded places, and adults have been found under tree bark in July (Hayat et al., 2002). Newly emerged females are in a state of reproductive dormancy, and although they mate, they are not able to lay eggs until completion of hibernation. In the spring, after feeding for 1–2 weeks, females lay a few eggs per day for 1–2 months before dying.

III. Affected Environment

A. Areas affected by yellow starthistle

Yellow starthistle is native to Eurasia, probably originating in the Mediterranean Region, and has spread to many temperate areas of the world, particularly those with Mediterranean climate (warm dry summer, cool rainy winter) (Maddox, 1981; Maddox et al., 1985). Its geographic distribution extends from Portugal and Spain in the west through France, Italy, Greece, the Balkans, and Turkey to Iran, Lebanon, Syria, Iraq, Azerbaijan, Kazakhstan, and southern Russia near the Black Sea in the east (Komarov, 1934; Klovov et al., 1963; Wagenitz, 1975; Dostál, 1976). More subspecies of yellow starthistle occur in or near Turkey than in any other region. Yellow starthistle occurs elsewhere in Europe and is naturalized in parts of central Europe (Dostál, 1976). In Eurasia, it occurs between 35° and 46°N latitude, especially at sunny disturbed sites in locations with a Mediterranean climate.

Yellow starthistle has spread to South Africa, Australia, Chile, and the United States. It has been reported in 41 of the 48 contiguous U.S. states, and it is listed as a noxious weed in 11 states and 2 Canadian provinces (Skinner et al., 2000; USDA-NRCS, 2008). However, it is most abundant in California, Oregon, Washington, and Idaho (Sheley et al., 1999; Duncan, 2001). A 1997 survey by the California Department of Food and Agriculture (CDFA) found yellow starthistle in 1,935 of California's 4,638 townships, and the infestations were reported as "high" in 1,019 of the townships (Pitcairn et al., 1998). In North America, yellow starthistle first appeared sometime after 1824, apparently arriving in contaminated shipments of alfalfa seed during the California gold rush in the 1850s (Gerlach, 1997a; b). In the 1870s and 1880s it spread to other Pacific West states, primarily by distribution of contaminated alfalfa seed. By 1900 it was a common weed in California, and by 1917 it had spread throughout California's Central Valley and was considered to be a serious

problem in grain fields. The weed has spread explosively during the last twenty years. A 2002 survey in California indicated that the area infested by the weed has increased by 81 percent since 1985. Yellow starthistle is spreading eastward into the Sierra Nevada mountains and over into Nevada and New Mexico. Colorado and Montana have eradication programs to try to prevent establishment of the weed in their states.

Yellow starthistle has adapted to a relatively wide range of environmental conditions and tolerates a variety of soil types (Maddox et al., 1985). In California, it is widely distributed in the central valleys and adjacent foothills and is currently spreading in mountainous regions up to 2,100 meters (7,000 feet). Yellow starthistle is less common in the desert, high mountains, and moister coastal areas. It is most abundant at sites with full sunlight and deep, well-drained soils where annual rainfall is between 25 to 150 millimeters (10–60 inches.). Habitats include rangelands, pastures, vineyards, abandoned croplands, wilderness areas, nature preserves, alfalfa and small grain fields, and roadsides. Yellow starthistle is found from flood plains and riverbanks to grasslands, ridges, and mountain slopes. It is most competitive in habitats that have a cool wet season that allows the rosettes time to send their roots down deep, followed by a warm-to-hot summer with little or no moisture that allows the plant to mature and produce seed with little interspecific competition. Yellow starthistle is particularly successful in the Mediterranean/Pacific-influenced climates of California, Oregon, and Washington and the intermontane grasslands of Idaho (Pitcairn et al., 1998; Piper, 2001; Prather et al., 2003).

B. Plants related to yellow starthistle and their distribution

Many plants in the family Asteraceae are worldwide in distribution, and relatives from the tribe Cardueae are commonly distributed throughout the range of yellow starthistle. In California, the federally and state listed Asteraceae are located along the central and southern California coast or in the Shasta River Valley in northern California. Artichoke (*Cynara scolymus*) is grown along the coast in central and southern California and in some agricultural regions of the interior desert. Safflower (*Carthamus tinctorius*) is grown in the San Joaquin and Sacramento Valleys and the intermountain areas of Washington, Idaho, Oregon, and Montana (Kaffka and Kearney, 2000). Sunflower (*Helianthus annuus*) is grown commercially in the Sacramento Valley of California and in the Northern Plains, especially North Dakota and Minnesota. Most, if not all of these species, overlap with the potential geographic range of yellow starthistle.

In North America, the subtribe Centaureinae includes *Carthamus tinctorius* (safflower), the weedy exotic *Centaurea* species (e.g., *Ce. maculosa*, *Ce. diffusa*, *Ce. melitensis*, *Ce. virgata* ssp. *squarrosa*), and two native *Centaurea* species (*Ce. americana* and *Ce. rothrockii*), which have recently been placed back in a separate genus (*Plectocephalus*). Bachelor's button (*Ce. cyanus*) is an introduced ornamental but is also considered a weed in many areas. Other widespread alien weeds in the subtribe Centaureinae include *Acroptilon repens* (Russian knapweed) and *Cnicus benedictus* (blessed thistle).

Related plants in other subtribes of the tribe Cardueae include the genera *Cirsium* and *Saussurea*, the cultivated artichoke (*Cynara scolymus*), and introduced weedy *Carduus* species. Other important commercial plants within the Asteraceae include lettuce (*Lactuca sativa*, tribe Lactuceae, subfamily Cichorioideae) and sunflower (*Helianthus annuus*, tribe Heliantheae, subfamily Asteroideae). Several important exotic ornamentals, including marigolds and asters, are also in the Asteraceae.

The North American *Cirsium* thistles represent the largest number of native plants closely related to yellow starthistle. Most native *Cirsium* spp. occur from the Rocky Mountains westward. Several species are becoming rare, and six species are federally listed as endangered or threatened: *Cirsium fontinale* var. *fontinale*, *Ci. loncholepis*, *Ci. vinaceum*, *Ci. fontinale* var. *obispoense*, *Ci. hydrophilum* var. *hydrophilum*, and *Ci. pitcheri*. A total of 20 native species, including *Ci. fontinale* ssp. *fontinale* and *Ci. fontinale* ssp. *obispoense*, occur in California. Eleven California species are considered rare (Tibor, 2001), and *Ci. ciliolatum* and *Ci. rhotophilum* are listed by the state of California as endangered and threatened, respectively.

The three native *Saussurea* species occur primarily in the Pacific Northwest, but one species, *S. americana*, has some minor populations in extreme northern California at high elevation (Moore and Frankton, 1973; Keil, 1993; Hitchcock and Cronquist, 1998).

IV. Environmental Consequences

A. No Action

1. Impact of yellow starthistle

Negative impacts of yellow starthistle

Yellow starthistle is a serious weed of pastures, rangelands, croplands,

roadsides, natural areas and recreational areas (Maddox et al., 1985). It is seldom a problem in heavily cultivated row crops but is sometimes troublesome in perennial crops such as hayfields, vineyards and orchards, and in dryland cereals (DiTomaso et al., 1998; Roché and Roché, 1988). It is an aggressive invader of disturbed sites and will continue to spread if left uncontrolled. It can become the dominant vegetation along roadsides, pastures and irrigation ditch banks. In pastures, yellow starthistle displaces desirable forage species and interferes with livestock grazing once the spines develop.

Consumption of yellow starthistle is toxic to horses (Kingsbury, 1964). Continued feeding causes ulcers in the mouth and results in brain lesions that cause a syndrome known as "chewing disease" or nigropallidal encephalomalacia (Cordy, 1978). There is no known treatment for horses that have been poisoned by yellow starthistle and in most cases the animals will die from starvation or dehydration (Panter, 1991). Repin, a sesquiterpene lactone, is thought to be the primary toxin (Merrill and Stevens, 1985; Hamburger et al., 1993). The highest risk of yellow starthistle poisoning is generally when the weed is the only green forage available or when it is a significant contaminant in dried hay. In some cases, horses acquire a taste for yellow starthistle and seek it out even when other forage is available (Panter, 1991). At least 100 horses were poisoned in northern California in 1954 (Cordy, 1954). Although other grazing animals do not appear to be susceptible to yellow starthistle's toxins, they are all vulnerable to eye damage caused by the plant's long sharp spines (Carlson et al., 1990).

Yellow starthistle also invades natural areas and displaces native plants, including natural wildlife forage. The spiny flowerheads reduce the use of recreational areas by the public. Yellow starthistle significantly depletes soil moisture reserves in annual grasslands in California (Dudley, 2000; Enloe et al., 2004) and in perennial grasslands in Oregon (Borman et al., 1992). Large yellow starthistle populations transpire the equivalent of about 4 to 8 inches of rainfall for each three feet of soil depth (Gerlach et al., 1998). Native perennial species such as blue oak (*Quercus douglasii*) and purple needlegrass (*Nassella pulchra*) depend on summer soil moisture reserves for growth and survival. However, because yellow starthistle uses deep soil moisture reserves earlier than these species, dense yellow starthistle infestations cause these plants to experience drought conditions even in years with normal rainfall (Gerlach et al., 1998). Yellow starthistle water consumption also impacts human economic interests (Dudley, 2000). The California State Water Resources Control Board acknowledged that control of weeds could significantly conserve water.

Ranchers are the demographic group most seriously affected by yellow starthistle. However, land managers, homeowners, outdoor enthusiasts, horse owners, and farmers also must contend with the harmful effects of this plant. Several public agencies are affected by yellow starthistle infestations at the federal, state, and county levels. These include the U.S. Bureau of Land Management, U.S. Forest Service, Bureau of Reclamation, U.S. Fish and Wildlife Service, Department of Defense, National Park Service, California Department of Fish and Game, California Department of Transportation, California Department of Food and Agriculture, California Department of Parks and Recreation, and County Agriculture Departments. Nongovernmental stakeholders include the California Cattleman's Association, wool growers, irrigation districts, the Nature Conservancy, the Audubon Society, the Sierra Club and the Farmland Trust.

Direct economic impact of yellow starthistle in California was recently estimated by the Agricultural Issues Center, University of California, Davis (Jetter et al., 2003) and is summarized by Smith (2006). Currently, yellow starthistle is considered to be well established and spreading exponentially. For this analysis it is estimated that 12 million acres are infested of a total 40 million acres that are susceptible. Assuming that the plant spreads at a rate of 10 percent per year, it is estimated that all susceptible lands will be infested within the next 10 years. Direct economic impact was estimated for two categories of land quality: high quality rangeland and degraded rangeland. Impact to high quality rangeland was estimated as the amount of land value lost following invasion by yellow starthistle. The estimate was derived from appraisals of ranches before and after they became infested, as provided by land appraisers. Such appraisals indicate that an infestation of yellow starthistle on high quality rangeland causes land values to decline by \$50 per acre. Appraisals of degraded land showed no difference between infested and uninfested properties. However, appraisers indicated that infested land took longer to sell than uninfested land. Average values for degraded rangeland were \$200 to \$300 an acre. At an annual interest rate of 7 percent, the interest would be approximately \$1.17 to \$1.75 if infested land took one month longer to sell. For this analysis, a more conservative estimate of \$1 an acre was assumed lost due to yellow starthistle. Total statewide impact for California was estimated as follows. Twelve million acres of land are infested with yellow starthistle and another 28 million acres are susceptible. If a benefit level of \$1 per acre is applied to both classes, total benefits for controlling yellow starthistle in California would be \$40 million. If a benefit level of \$1 per acre is applied to infested sites and \$50 per acre to land susceptible to yellow starthistle, total benefits are \$1.412 billion.

It should be noted that the above land valuation approach primarily captures benefits from changes in rangeland productivity, improved land access, changes in weed management, and enhanced aesthetics. It does not include environmental benefits such as reduced exposure to herbicides, increased biodiversity, or increased availability of water to recreation, agriculture, and native vegetation. For example, one study has estimated that water consumption by yellow starthistle is worth \$16 to \$75 million per year in the Sacramento River watershed alone (Gerlach, 2004). Although such estimates have not been made on a statewide or national basis, these additional costs are clearly substantial.

Benefits of yellow starthistle

Yellow starthistle has some benefits. Some beekeepers value it as a source of late-season pollen and nectar when most other flowers have disappeared (Edwards, 1989; Goltz, 1999). In 1959, about 150,000 bee colonies used yellow starthistle as a source of pollen and nectar, which produced honey worth between \$150,000 and \$200,000 (Maddox et al., 1985). On the other hand, domesticated honey bees are themselves aliens, and they compete with native bees in natural habitats for pollen sources. Since many native pollinators are specialized for their host plants, yellow starthistle may actually encourage displacement of the native bees by honey bees. In a study conducted on Santa Cruz Island in California, investigators found that honeybees visited yellow starthistle 33 times more than native bees, whereas native bees visited a native gumplant species (*Grindelia camporum*) 46 times more than honey bees (Barthell et al., 2000). Yellow starthistle also displaces native flowers to which native pollinators are adapted.

Young yellow starthistle plants have some forage value to cattle before they bolt and develop spines (Callihan et al., 1995). Young plants contain between 8 to 14 percent protein (Thomsen et al., 1990), but cattle subsisting on the plant lose weight (Callihan et al., 1982). The flat-lying rosettes are also difficult for cattle to bite. The total forage in yellow starthistle-infested fields has less value to cattle than that in uninfested fields, based on consumption of crude protein and total digestible nutrients (Barry, 1995).

Yellow starthistle produces seed that is consumed by some birds (including ring-necked pheasants, mourning doves, California quail, gold finches, and house finches) and rodents (Martin et al., 1951; Roché, 1965). However, seed produced by yellow starthistle is at the expense of that produced by native forbs and grasses which are displaced by the weed. If other wildlife such as deer were found to graze on yellow starthistle rosettes, it is similarly likely that the weed would not provide better forage

than native plants, especially when the overabundance of the invasive alien weed species limits the variety of plants available to wildlife. In any case, when birds feed on yellow starthistle seed, some viable seed can pass through their guts, which increases the risk of dispersing the weed to new locations (Roché, 1965).

2. Impact from use of other control methods

The continued use of chemical herbicides and mechanical, cultural, and biological controls at current levels would be a result if the “no action” alternative is chosen.

Chemical control

Resistance of yellow starthistle to herbicides was first observed in 1989 in Dayton, Washington. This population was resistant to picloram and cross-resistant to clopyralid, dicamba, and fluroxypyr that have a similar mode of action as picloram (Callihan et al., 1990). The development of picloram-resistant yellow starthistle indicates the potential for development of resistance to clopyralid if the herbicide is used year after year. Controlling resistant populations requires using higher concentrations of herbicide, using other less desirable herbicides, developing new herbicides, or else abandoning herbicides as a management tool.

Continuous use of herbicides may cause one noxious weed to be replaced by another equally undesirable species that is less sensitive to the herbicide. When broadleaf-selective herbicides are used, noxious annual grasses such as medusahead (*Taeniatherum caput-medusae*), cheatgrass or downy brome (*Bromus tectorum*), or barbed goatgrass (*Aegilops triuncialis*) may become dominant. Repeated use of clopyralid may also reduce plant diversity, especially of legumes, and decrease range productivity (DiTomaso, 1997).

Mechanical control

Hand pulling and hoeing disturb the soil and create an ideal site for re-establishment of new seedlings or rapid invasion of other weeds (DiTomaso, 1997). Tillage can be effective, but is not appropriate for rangeland (Thomsen et al., 1996). Some disadvantages of mowing are that it must be done during a narrow time window in order to be effective (Benefield et al., 1999), it can interfere with the seedhead insect biological control agents, injure late growing native forb species, and reduce fall and winter forage for wildlife and livestock (DiTomaso, 1997; DiTomaso et al., 2000).

Cultural control

Fire has little if any impact on seeds in the soil. Burning can increase plant size and seed production of yellow starthistle in the following year by releasing nutrients otherwise tied up in the plant material and by removing shading thatch and vegetation. Burning also can increase soil erosion. Burning requires a high number of trained personnel to manage the fire and keep it contained, permits, and proper weather conditions. This makes the technique difficult and expensive to implement over large areas. Environmental conditions, the risk of wildfire, and air quality regulations often restrict the use of this practice. Finally, burning alone does not eradicate yellow starthistle, but it can lead to a temporary increase in native and desirable vegetation (DiTomaso et al., 1999). Successful control depends on follow-up management, and a second year burn is usually not practical because of insufficient thatch for fuel. However, very good control can be achieved by using prescribed burning followed the next year by a clopyralid treatment (DiTomaso et al., 2003).

Grazing cattle in late winter or early spring can harm desirable grasses and increase yellow starthistle infestations. Trampling vegetation also tends to favor yellow starthistle (Miller et al., 1998). Intensive grazing requires frequent movement of livestock, and may employ movable electric fencing, both of which are relatively labor intensive. Although goats continue to browse yellow starthistle even in the flowering stage (Thomsen et al., 1993), when confined they can damage both desirable and undesirable species and may even strip the bark off trees.

When successful, revegetation with desirable and competitive plant species can be the best long-term sustainable method of suppressing invasive weeds, while providing high forage production. Unfortunately, it is often difficult to establish the desired species. Seed of native species is usually not available in large quantities, is expensive, and establishment is often difficult to achieve. Furthermore, use of native seed from one area in another area can be controversial.

Biological Control

Surveys conducted in California show that each of the flowerhead insects are most abundant in different environmental regions (Pitcairn et al., 2003). This limits the effectiveness of each species, but *E. villosus* and *Ch. succinea* tend to do best in complementary environments, the former in cooler, hilly regions and the latter in hotter parts of the Central Valley.

Post-release field impact studies of the established insect biological

control agents at three sites in California have shown that the insects attack about 50 to 90 percent of flowerheads, depending on year and site. At some sites yellow starthistle seed production and density of adult plants have generally tended to decrease over a 7 year period (Pitcairn et al., 2003; Woods et al., 2004b). Some sites in Oregon with high densities of flowerhead insects and exclusion of cattle grazing and soil disturbance have also experienced decreases in yellow starthistle density.

Although infection occurred at all sites, the rust, *P. jaceae* var. *solstitialis*, has not yet shown signs of spreading very far. In California where, after inoculations at 176 sites in 40 counties, it has become permanently established only in Sonoma Co., near San Pablo Bay (Woods et al., 2010). *Puccinia jaceae* var. *solstitialis* was released in Oregon between May 2008 and April 2011 and has established near Myrtle Creek (Bruckhart et al., 2016). The rust is expected to reduce the size and seed production of yellow starthistle but not to cause much plant mortality (Shishkoff and Bruckart, 1996).

These environmental consequences may occur even with the implementation of the biological control alternative, depending on the efficacy of *C. basicorne* to reduce yellow starthistle populations.

B. Issue Permits for Environmental Release of *Ceratapion basicorne*

1. Impact of *C. basicorne* on non-target plants

No-choice results indicate that no plant species outside the subtribe Centaureinae are at risk of larval damage (Smith, 2007 – appendix 2). There is no risk of larval damage to any native North American plant species, although there may be a small risk of adult feeding and egg laying on *Saussurea americana*, a rare plant occurring in northern California at a high elevation (1,700 meters), but is relatively widespread in the Pacific Northwest. This plant prefers damp cool habitats and generally does not occur near heavy infestations of yellow starthistle. This geographic isolation is likely to further reduce any risk to this plant, because it would not occur near high densities of the insect. The closest native species to yellow starthistle are *Ce. americana* and *Ce. rothrockii*, but they are so distantly related as to be considered in a different genus (*Plectocephalus*), and they were not able to maintain larval development in no-choice experiments (Smith, 2007 – see appendix 2).

The laboratory choice and no-choice experiments indicate that *Ce. cyanus* (bachelor's button) is at some risk of adult feeding, egg laying, and larval

damage (Smith, 2007 - see appendix 2). This plant has previously been reported as a host of *C. basicorne* in its native range in Eurasia, but the frequency of such attack has not been studied. The plant is considered both an ornamental and invasive in parts of North America. Although this may cause cosmetic damage to the ornamental, it is not known how frequently this will occur, especially when the plant is grown in an intensely managed monoculture (Smith, 2007). However, because this plant is an invasive weed in some regions of the western United States, damage to it in rangeland habitats would be beneficial (Smith, 2007 – see appendix 2).

The results also indicate that there may be low attack and larval damage to *Ce. melitensis* (tocalote), which is an invasive alien weed. Two other invasive alien weeds, *Crupina vulgaris* and *Cnicus benedictus*, can also support larval development, but risk of attack was not measured in choice experiments because there is no interest to protect either species in North America. It should be noted that *Crupina vulgaris* has been considered as a potential target for classical biological control (Roché et al., 2003).

Ceratapion basicorne is physiologically capable of developing on safflower. Laboratory choice experiments showed a low egg laying rate on some varieties of this plant under confined conditions. However, during three years of field studies in eastern Turkey, no specimens of *C. basicorne* were reared from safflower. At two sites where *C. basicorne* was the only apionid species present, safflower was not infested by any internal feeding insect, despite infestation rates of 48 to 98 percent in yellow starthistle. At another location where *C. scalptum*, *C. orientale*, and *C. onopordi* were present, 8 to 34 percent of safflower plants were infested, but of 19 identifiable insects reared from safflower during 3 years, none were *C. basicorne*.

Ceratapion basicorne has been reared in the field from only *Ce. solstitialis* (yellow starthistle), *Ce. cyanus* (bachelor's button), *Ce. depressa*, and *Cnicus benedictus* (blessed thistle). Molecular genetic research indicates that "*Cnicus*" belongs in the genus *Centaurea* and that it is closely related to *Ce. solstitialis* (Garcia-Jacas et al., 2001). Thus, *C. basicorne* has been reared from only four species of field-collected plants, all of which are in the genus *Centaurea*. This insect has never been reported as a pest of safflower in Eurasia, where the insect naturally occurs (Bytinski-Salz, 1952; Avidov and Kotter, 1966). It should be noted that, in general, field tests (measuring ecological host range) have more accurately predicted risk to nontarget plants than no-choice laboratory experiments (measuring physiological host range) (Clement and Cristofaro, 1995; Briese, 2005; Sheppard et al., 2005). Under field conditions, *C. basicorne* does not damage safflower, and that with respect to this crop, the insect is safe to

introduce to North America (Smith et al., 2006; Cristofaro et al., 2013).

2. Impact of *C. basicorne* on yellow starthistle

In a quarantine laboratory experiment, infestation by weevil larvae caused up to a 23 percent reduction in size of well-watered, fertilized potted plants grown in a greenhouse. The mean number of insects per plant was 3.1 (\pm 1.5 standard deviation) in the low infestation treatment and 4.2 (\pm 3.0 standard deviation) in the high infestation treatment. In the field in Turkey, up to seven larvae have been found in yellow starthistle roots (Uygur et al., 2005). If this insect attains high densities typical for a successfully established biological control agent, it is likely to cause more damage than observed in the quarantine impact experiment. In the field, where plants compete for water, nutrients, and light, weevil infestation may have greater impact on plant size, survivorship, and reproduction. Furthermore, adult feeding on plant leaves in the spring may cause significant damage if weevil populations become very large, as was observed for *Larinus minutus* on diffuse knapweed (Piper, 2004). In a field study of naturally occurring yellow starthistle plants in Turkey, plants infested by *Ceratapion* had 15 percent lower seed fertility than uninfested plants (Uygur et al., 2005). Although it is difficult to predict how much impact *C. basicorne* will have on yellow starthistle populations in North America, it is expected that the weevil has potential to affect the plant. Furthermore, because *C. basicorne* feeds on the rootcrown and leaves, it is not likely to directly interfere with the previously released flowerhead insects that do not feed on these plant parts (Smith, 2006).

3. Uncertainties regarding the environmental release of *C. basicorne*

Once a biological control agent such as *C. basicorne* is released into the environment and becomes established, there is a slight possibility that it could move from the target plant (yellow starthistle) to attack nontarget plants. Host shifts by introduced weed biological control agents to unrelated plants are rare (Pemberton, 2000). Native species that are closely related to the target species are the most likely to be attacked (Louda et al., 2003). If other plant species were to be attacked by *C. basicorne*, the resulting effects could be environmental impacts that may not be easily reversed. Biological control agents such as *C. basicorne* generally spread without intervention by man. In principle, therefore, release of this biological control agent at even one site must be considered equivalent to release over the entire area in which potential hosts occur, and in which the climate is suitable for reproduction and survival. However, significant

non-target impacts on plant populations from previous releases of weed biological control agents are unusual (Suckling and Sforza, 2014).

In addition, these agents may not be successful in reducing yellow starthistle populations in the continental United States. Worldwide, biological weed control programs have had an overall success rate of 33 percent; success rates have been considerably higher for programs in individual countries (Culliney, 2005). Actual impacts on yellow starthistle by *C. basicorne* will not be known until after release occurs and post-release monitoring has been conducted (see appendix 2 for post-release monitoring plan).

4. Cumulative impacts

“Cumulative impacts are defined as the impact on the environment which results from the incremental impact of the action when added to other past, present and reasonably foreseeable future actions regardless of what agencies or person undertakes such other actions” (40 CFR 1508.7).

Many states and counties, particularly in the western United States, conduct weed control programs to manage yellow starthistle as well as other invasive weeds. Chemical, mechanical, and biological controls, as described previously in this document are used in a wide range of habitats.

Release of *C. basicorne* is not expected to have any negative cumulative impacts in the continental United States because of its host specificity to yellow starthistle. Effective biological control of yellow starthistle will have beneficial effects for weed management programs, and may result in a long-term, non-damaging method to assist in the control of yellow starthistle, and prevent its spread into other areas potentially at risk from invasion.

5. Endangered Species Act

Section 7 of the Endangered Species Act (ESA) and its implementing regulations require Federal agencies to ensure that their actions are not likely to jeopardize the continued existence of federally listed threatened endangered species or result in the destruction or adverse modification of critical habitat.

Ceratapion basicorne is not expected to directly affect any plants outside the tribe Cardueae (thistles) in the Asteraceae family. Some adult feeding and egg laying on leaves may occur on some species of plants in the Cardueae, especially those in the subtribe Centaureinae during periods when weevil densities are high in relation to available yellow starthistle

plants (e.g., temporary "spillover" effect during a rapid decrease of the target weed population). Choice and no-choice experiments indicate that no native North American plant species are at risk of significant larval damage from this weevil. Root or stem damage caused by developing larvae was observed only on species in the subtribe Centaureinae (no listed species occur in this subtribe). In the field, larval damage may be expected to occur on bachelor's button (*Centaurea cyanus*), tocalote (*Ce. melitensis*), common crupina (*Crupina vulgaris*), and blessed thistle (*Cnicus benedictus*), all non-native species. This damage is most likely to occur during the transitory period when yellow starthistle populations decrease in the presence of high densities of weevils.

APHIS has determined that environmental release of the weevil *Ceratapion basicorne* may affect, but is not likely to adversely affect *Cirsium fontinale* var. *fontinale*, *Ci. loncholepis*, *Ci. vinaceum*, *Ci. fontinale* var. *obispoense*, *Ci. hydrophilum* var. *hydrophilum*, and *Ci. pitcheri* or their critical habitats. This determination is based on host specificity testing where eggs were laid on the plant and/or adult feeding holes were present on plants. Although eggs were laid on some plants in no-choice testing, no larval development occurred on any threatened or endangered plants, and adult feeding on foliage during no-choice tests was minor. In additional testing conducted on *Cirsium* spp., the amount of feeding damage to *Cirsium* species in the no-choice experiment was extremely small, always less than 0.1 percent of the leaf surface area. The highest rate of feeding on any *Cirsium* species observed in the no-choice experiment was 4.0 holes per day on *Cirsium loncholepis*, which is equivalent to 0.089 percent of one leaf, or 0.009 percent of the total leaf area on a young rosette. However, this insect is not attracted to *Ci. loncholepis*, nor has this insect ever been collected on any other *Cirsium* species (Alonso-Zarazaga, 1990; Wanat, 1994). Adult feeding is inconsequential even on yellow starthistle and has no impact on that plant.

APHIS submitted a biological assessment to the U.S. Fish and Wildlife Service (FWS) requesting concurrence with its determination that environmental release of *C. basicorne* was not likely to adversely affect listed *Cirsium* species in the continental United States. APHIS received a letter from FWS (dated May 22, 2008), concurring with this determination. No new *Cirsium* species have been listed since then.

V. Other Issues

Consistent with Executive Order (EO) 12898, "Federal Actions to Address Environmental Justice in Minority Populations and Low-income Populations," APHIS considered the potential for disproportionately high

and adverse human health or environmental effects on any minority populations and low-income populations. There are no adverse environmental or human health effects from the field release of *C. basicorne* and will not have disproportionate adverse effects to any minority or low-income populations.

Consistent with EO 13045, “Protection of Children From Environmental Health Risks and Safety Risks,” APHIS considered the potential for disproportionately high and adverse environmental health and safety risks to children. No circumstances that would trigger the need for special environmental reviews is involved in implementing the preferred alternative. Therefore, it is expected that no disproportionate effects on children are anticipated as a consequence of the field release of *C. basicorne*.

EO 13175, “Consultation and Coordination with Indian Tribal Governments,” was issued to ensure that there would be “meaningful consultation and collaboration with tribal officials in the development of Federal policies that have tribal implications....” In August 2008, APHIS sent out letters to tribal leaders and organizations of six states (Washington, Oregon, California, Nevada, Arizona, and Idaho) to give notification of the proposed environmental release of *C. basicorne* and to request input from tribes. APHIS will continue to consult and collaborate with Indian tribal officials to ensure that they are well-informed and represented in policy and program decisions that may impact their agricultural interests, in accordance with EO 13175.

VI. Agencies, Organizations, and Individuals Consulted

The Technical Advisory Group for the Biological Control Agents of Weeds (TAG) recommended the release of *C. basicorne* on September 15, 2006. TAG members that reviewed the release petition (Smith, 2006) included representatives from the U.S. Fish and Wildlife Service, Bureau of Indian Affairs, Weed Science Society of America, Cooperative State Research, Education, and Extension Service, U.S. Geological Survey, Environmental Protection Agency, U.S. Forest Service, U.S. Army Corps of Engineers, Bureau of Reclamation, Bureau of Land Management, the National Plant Board, Animal and Plant Health Inspection Service, and Agriculture and Agri-Food Canada, Health Canada.

This EA was prepared and reviewed by APHIS and ARS. The addresses of participating APHIS units, cooperators, and consultants (as applicable) follow.

U.S. Department of Agriculture
Animal and Plant Health Inspection Service
Policy and Program Development
Environmental and Risk Analysis Services
4700 River Road, Unit 149
Riverdale, MD 20737

U.S. Department of Agriculture
Animal and Plant Health Inspection Service
Pests, Pathogens, and Biocontrol Permits
4700 River Rd., Unit 133
Riverdale, MD 20737

U.S. Department of Agriculture
Agricultural Research Service
Exotic Invasive Weeds Research Unit
Western Regional Research Center
800 Buchanan Street
Albany, CA 94710

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Appendix 1. Host Plant Test List (all in family Asteraceae).

No.	Tribe	Subtribe	Test Species	Notes
Subfamily Cichorioideae				
1	Arctoteae		<i>Gazania rigens</i> (L.) Gaertn.	introduced ornamental
2	Cardueae	Centaureinae	<i>Centaurea americana</i> Nutt.	native
3	Cardueae	Centaureinae	<i>Centaurea calcitrapa</i> L.	introduced weed
4	Cardueae	Centaureinae	<i>Centaurea cineraria</i> L.	introduced ornamental
5	Cardueae	Centaureinae	<i>Centaurea cyanus</i> L.	introduced ornamental/weed
6	Cardueae	Centaureinae	<i>Centaurea dealbata</i> Willd.	introduced
7	Cardueae	Centaureinae	<i>Centaurea diffusa</i> Lam.	introduced weed
8	Cardueae	Centaureinae	<i>Centaurea debeauxii</i> Gren. & Godr. ssp. <i>thuillieri</i> Dostál (=C. x <i>pratensis</i>)	introduced weed
9	Cardueae	Centaureinae	<i>Centaurea maculosa</i> Lam.	introduced weed
10	Cardueae	Centaureinae	<i>Centaurea melitensis</i> L.	introduced weed
11	Cardueae	Centaureinae	<i>Centaurea montana</i> L.	introduced ornamental
12	Cardueae	Centaureinae	<i>Centaurea rothrockii</i> Greenm.	native
13	Cardueae	Centaureinae	<i>Centaurea solstitialis</i> L.	target
14	Cardueae	Centaureinae	<i>Centaurea sulphurea</i> Willd.	introduced weed
15	Cardueae	Centaureinae	<i>Centaurea virgata</i> Lam. var. <i>squarrosa</i> (Willd.) Boiss.	introduced weed
16	Cardueae	Centaureinae	<i>Carthamus tinctorius</i> L., var. CW-88 OL <i>Carthamus tinctorius</i> L., var. CW-1221 <i>Carthamus tinctorius</i> L., var. CW-4440 <i>Carthamus tinctorius</i> L., var. Gila <i>Carthamus tinctorius</i> L., var. Hartman <i>Carthamus tinctorius</i> L., var. S-400 <i>Carthamus tinctorius</i> L., var. S-541	safflower, CalWest, oleic safflower, CalWest safflower, CalWest, linoleic safflower (Arizona) safflower, linoleic (Montana) safflower, SeedTec safflower, SeedTec, linoleic
17	Cardueae	Centaureinae	<i>Acroptilon repens</i> (L.) DC.	introduced weed
18	Cardueae	Centaureinae	<i>Cnicus benedictus</i> L.	introduced weed
19	Cardueae	Centaureinae	<i>Crupina vulgaris</i> Cass.	introduced weed
20	Cardueae	Carduinae	<i>Carduus pycnocephalus</i> L.	introduced weed
21	Cardueae	Carduinae	<i>Cirsium brevistylum</i> Cronq.	native
22	Cardueae	Carduinae	<i>Cirsium ciliolatum</i> (Henderson) Howell	native
23	Cardueae	Carduinae	<i>Cirsium cymosum</i> (Greene) Jepson	native
24	Cardueae	Carduinae	<i>Cirsium fontinale</i> Greene var. <i>fontinale</i>	native
25	Cardueae	Carduinae	<i>Cirsium hydrophilum</i> (Greene) Jepson var. <i>vaseyi</i> (Gray) Howell	native
26	Cardueae	Carduinae	<i>Cirsium loncholepis</i> Petrak	native
27	Cardueae	Carduinae	<i>Cirsium occidentale</i> (Nutt.) Jepson var. <i>venustum</i> (Greene) Jepson	native

28	Cardueae	Carduinae	<i>Cirsium vulgare</i> (Savi) Ten.	introduced weed
29	Cardueae	Carduinae	<i>Cirsium vinaceum</i> Woot. & Standl.	native
	Tribe	Subtribe	Test Species	Notes
30	Cardueae	Carduinae	<i>Cynara scolymus</i> L.	artichoke, Globe
31	Cardueae	Carduinae	<i>Saussurea americana</i> Eaton	native
32	Cardueae	Carduinae	<i>Onopordum acanthium</i> L.	introduced weed
33	Cardueae	Carduinae	<i>Silybum marianum</i> (L.) Gaertn.	introduced weed
34	Cardueae	Carlininae	<i>Xeranthemum cylindraceum</i> Sibth. & Sm.	introduced ornamental
35	Cardueae	Echinopsidinae	<i>Echinops exaltatus</i> Schrad.	introduced ornamental
36	Mutiseae		<i>Trixis californica</i> Kellogg	native
37	Lactuceae		<i>Agoseris grandiflora</i> (Nutt.) Greene	native
38	Lactuceae		<i>Lactuca sativa</i> L.	crop, lettuce
39	Lactuceae		<i>Stephanomeria cichoriacea</i> Gray	native
40	Vernonieae		<i>Stokesia laevis</i> (Hill) Greene	native, ornamental
Subfamily Asteroideae				
41	Anthemideae		<i>Artemisia californica</i> Less.	native
42	Astereae		<i>Symphyotrichum</i> (=Aster) <i>chilense</i> (Nees) Nesom var. <i>chilense</i>	native
43	Eupatorieae		<i>Brickellia californica</i> (Torr. & Gray) Gray	native
44	Eupatorieae		<i>Liatrix punctata</i> Hook.	native
45	Gnaphalieae		<i>Pseudognaphalium</i> (=Gnaphalium) <i>californicum</i> (DC.) A. Anderb.	native
46	Helenieae		<i>Eriophyllum stoechadifolium</i> Lag.	native
47	Helenieae		<i>Hemizonia minthornii</i> Jepson	native
48	Heliantheae		<i>Echinacea purpurea</i> (L.) Moench	native
49	Heliantheae		<i>Helianthus annuus</i> L.	native, crop, sunflower
50	Plucheeae		<i>Pluchea odorata</i> (L.) Cass. ¹	native
51	Senecioneae		<i>Senecio bicolor</i> (Willd.) Todaro ssp. <i>cineraria</i> (DC.) Chater	introduced ornamental
52	Senecioneae		<i>Senecio vulgaris</i> L.	introduced weed

Appendix 2. Protocol for Releasing *C. basicorne* and Post-Release Monitoring Plan

Protocol for Releasing *C. basicorne*

It is critical to be certain that all individuals that would be released are correctly identified to avoid the accidental release of other species. *Ceratapion basicorne* and *C. orientale* are the only *Ceratapion* species that have been reared from yellow starthistle (Alonso-Zarazaga, 1990a; Wanat, 1994; Smith et al., 2006). *Ceratapion scalptum*, which attacks safflower, has never been reared from yellow starthistle, so the risk of accidentally introducing this species can be minimized by using only insects reared from yellow starthistle. The permittee plans to release individuals that are descendents from the colony in the USDA-ARS quarantine laboratory that was used to conduct the experiments reported here. This population was established by rearing adults from yellow starthistle plants infested with apionid larvae and pupae that were collected at sites near Kayseri, Sivas, Erzincan, Erzurum, and Malatya, Turkey between June 4 and 8, 2001. If additional individuals should be needed, more individuals will be collected in the same way, from the same region and reared in quarantine to produce progeny for field release after all parents have been identified by a recognized weevil taxonomist. Voucher specimens will be kept at the USDA-ARS laboratory in Albany, CA and others will be deposited at the California Department of Food and Agriculture in Sacramento, CA, and at the USDA Systematic Entomology Laboratory (SEL) in Beltsville, MD.

Adult insects that have completed hibernal diapause will be released inside large cages (3 x 3 x 2 m) in the spring in dense patches of yellow starthistle at experimental sites. Experimental sites will be selected in several counties in California that represent different climatic zones (Central valley, Coastal Range and Sierra Foothills) where yellow starthistle is common. Specific sites will be selected in cooperation with California Dept. of Agriculture (CDFA), County Depts. of Agriculture, California Dept. of Transportation and other interested agencies. During the initial releases, experiments will be conducted to compare the success of establishment as a function of the number of female beetles released in cages (5, 10, 20). This procedure is similar to those used by others to successfully release weevil weed biological control agents in the United States (Story et al., 1997; Villegas et al., 2000; Coombs, 2004). After data on establishment and impact on target and nontarget plants have been analyzed, secondary releases will be made in California, Oregon, Idaho, Washington, Nevada and possibly in other states, depending on the demand for and availability of insects. When weevil populations become well established at the release sites, they will be harvested for release at secondary sites.

Post-Release Monitoring

At the release sites, the permittee will monitor establishment and increase of the weevil population inside the release cages. Impact of the weevil on yellow starthistle survival, plant size, and production of viable seed will be measured by comparing infested to

uninfested plants at the release sites. After weevil progeny emerge in early summer, bolting yellow starthistle plants will be dug up and dissected to determine the attack rate and impact of the insect will be measured by comparing infested plants to nearby uninfested plants. Research will be conducted to identify pheromones that could be used in traps to use in future studies to monitor the spread of the weevil.

Appendix 3. Smith, L. 2007. Physiological host range of *Ceratapion basicorne*, a prospective biological control agent of *Centaurea solstitialis* (Asteraceae). *Biological Control*. 41: 120–133.

Physiological host range of *Ceratapion basicorne*, a prospective biological control agent of *Centaurea solstitialis* (Asteraceae)

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Abstract

Ceratapion basicorne (Coleoptera: Apionidae) is a weevil native to Europe and western Asia that is being evaluated as a prospective classical biological control agent of *Centaurea solstitialis* (yellow starthistle) in the United States. Host plant specificity of the insect was evaluated in no-choice oviposition experiments. Feeding on leaf tissue by adult females was highly correlated to oviposition rate, both of which occurred primarily on plants in the tribe Cardueae, and especially those in the monophyletic subtribe Centaureinae. The highest rates of larval development occurred on *Ce. solstitialis* and *Centaurea cyanus* (bachelor's button, garden cornflower), and there was significant development on *Centaurea melitensis* (Napa starthistle, tocalote), *Cnicus benedictus* (blessed thistle), *Carthamus tinctorius* (safflower), and *Crupina vulgaris* (common crupina). All the plants that supported some larval development are within a monophyletic clade within the Centaureinae. No native North American plants appear to be at risk of significant damage by this insect. Additional testing of safflower and bachelor's button under choice conditions should complement these results to help determine the degree to which these plants are at risk.
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Keywords: Host plant specificity; Biological control; Weed; Invasive plant; Risk assessment

1. Introduction

1.1. Weed distribution, ecology, and impact

Yellow starthistle (*Centaurea solstitialis* L.) is an invasive alien weed that was accidentally introduced into California over 130 years ago, primarily by importation of contaminated alfalfa seed (Maddox et al., 1985; Gerlach, 1997a,b). The weed infests about eight million hectares in the western US and Canada (Duncan, 2001; Pitcairn et al., 2006). Infestations have been reported in 23 states, with the largest populations in the states of California, Idaho, Oregon, and Washington (Maddox et al., 1985; Sheley et al., 1999). It is considered the most common weed in California, and it is continuing to spread and threaten states to the east (Pitcairn et al., 2006). The weed is designated as noxious in 11 western states and two Canadian provinces (Skinner et al., 2000).

Yellow starthistle is highly invasive in grassland habitats and displaces desirable plants in both natural and grazing areas. Spiny flowerheads interfere with grazing animals and human recreation, and the plants displace desirable vegetation and deplete soil moisture. Consumption of yellow starthistle by horses causes a fatal syndrome known as “chewing disease” or nigropallidal encephalomalacia (Cordy, 1978). Total economic benefits for controlling yellow starthistle in California have been estimated to be between \$40 million and \$1.4 billion, depending on assumptions (Jetter et al., 2003). Environmental benefits due to reduced use of herbicides, increased recreation and increased biodiversity have not been estimated, nor have any benefits to nearby states.

Yellow starthistle is an herbaceous winter annual plant native to southern Europe and the Near East (Maddox, 1981), occurring from Spain to Iran (Wagenitz, 1975; Dostál, 1976; Rechinger, 1980). The geographic center of origin may be in Turkey or Greece, based on the number of subspecies occurring in these regions (Wagenitz, 1975; Dostál, 1976). Although the plants in the western USA are genetically

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diverse, there is no evidence of genetically distinct subpopulations occurring there (Roché, 1965; Sheley et al., 1983a,b; Schumacher et al., 1982; Sun, 1997). The plant is well adapted to a Mediterranean climate (dry summers and wet winters) and can tolerate winter snow. In California, seeds germinate mainly in early winter, rosettes grow slowly until spring, then the plants “bolt” and bloom until they die from desiccation or frost (Pitcairn et al., 1999a; Sheley et al., 1999). Some seeds are released during the summer as individual capitula (flower heads) mature while others are retained until the capitula disintegrate during winter. Seeds falling in summer soon become dormant and require cooler temperatures before germinating, usually soon after the start of winter rains. Seeds buried in soil can remain viable for several years and will germinate after exposure to light when the soil is disturbed (Joley et al., 2003 and Refs. therein).

1.2. Weed management options

Although several herbicides are effective (Sheley et al., 1999; DiTomaso, 2005), conventional herbicide control strategies have often been inadequate because of the large areas infested, the economic and environmental costs of herbicides, or the relatively low monetary return from land used for grazing, rights-of-way, conservation or recreation. Other control strategies such as tillage, mowing, burning, and grazing have been evaluated and can sometimes be effective, but are not practical for managing the weed over large areas of rangeland (DiTomaso et al., 2000; DiTomaso, 2005 and Refs. therein). In the Mediterranean region, where it originates, yellow starthistle generally occurs in low densities and appears to be under natural control (Uygur et al., 2004). Research to discover, evaluate, and introduce classical biological control agents began in the 1960s (Maddox, 1981; Rosenthal et al., 1992; Turner et al., 1995; Sheley et al., 1999). Some biological control agents have been previously introduced, with the establishment of six exotic insect species, all of which attack flowerheads and destroy developing seeds (Turner et al., 1995; Balciunas and Villegas, 2001). A few indigenous invertebrates and diseases have been described on yellow starthistle in California (Pitcairn et al., 1999a,b). Of these, only a few attack plants later than the seedling stage, and these have little effect (Klisiewicz, 1986), but some can cause significant mortality among very young seedlings (Woods et al., 2000). The rust pathogen, *Puccinia jaceae* var. *solstitialis*, was introduced in California in 2003 (Woods et al., 2004; Fisher et al., 2006), but it does not appear to be causing significant damage to the plant. The combined effect of these natural enemies has not significantly reduced yellow starthistle over most of its range (Balciunas and Villegas, 1999; Pitcairn et al., 2002; Smith, 2002), although there are some local reductions, especially in the presence of competing vegetation (Pitcairn et al., 2005; E.M. Coombs, personal communication). Comparative life history studies of the plant in California (Pitcairn et al., 2002) and Turkey (Uygur et al., 2004) suggest that natural enemies that damage the rosettes may be most

effective for controlling it. Additional agents are needed, especially ones that attack the foliage, stem, and roots of rosettes and young bolting plants (Smith, 2004).

1.3. Life history and behavior of *Ceratapion basicorne*

Ceratapion basicorne (Illiger) (Coleoptera: Apionidae) is a weevil native to Europe and the Near East that develops in rosettes of yellow starthistle (Clement et al., 1989; Alonso-Zarazaga, 1990a; Wanat, 1994). In the wild this insect has been reared from *Ce. solstitialis*, *Ce. cyamus* L., and *Cnicus benedictus* L., which suggests that it is highly host-specific (Alonso-Zarazaga, 1990a; Wanat, 1994; Campobasso et al., 1999). The insect is common on yellow starthistle in Turkey, Greece, and Georgia (Rosenthal et al., 1994; Balciunas, 1998; Uygur et al., 2005) and is widely distributed in Europe and western Asia (Alonso-Zarazaga, 1990a; Wanat, 1994). Overwintering adults become active in the early spring and feed on rosette leaves (Clement et al., 1989). Eggs are deposited inside leaves, and larvae tunnel down the leaf petiole and develop inside the upper root and basal stem (root crown), where they pupate (Smith and Drew, 2006). Females oviposit about 1.5 eggs per day during an oviposition period of about 20 days. Development time of eggs until eclosion of larvae is 8.5 days at room temperature (19°C), and development time from oviposition until adult emergence is about 77 days. Adults emerge from the plant in early summer, when it bolts. Adults feed briefly on yellow starthistle foliage then aestivate and hibernate until the following spring. Mating occurs soon after new adults emerge in early summer and after hibernation ends in the following spring (Smith and Drew, 2006). There is one generation per year.

Some data on host specificity were reported by Clement et al. (1989), indicating that *Carthamus tinctorius* L. (safflower), *Galactites tomentosus* Moench, and *Carduus pycnocephalus* L. can support larval development. However, absence of reports of the development of this insect from these hosts in the field (Alonso-Zarazaga, 1990a; Wanat, 1994; Campobasso et al., 1999; Uygur et al., 2005) prompted me to further evaluate its host specificity. The purpose of the present study was to determine what non-target plants are susceptible to damage by *C. basicorne* and to measure the relative amount of damage the insect can cause under no-choice laboratory conditions. Such data provide part of the basis for assessing the risk that the insect would pose to non-target plants if it were to be introduced as a biological control agent (USDA-APHIS, 1998; Withers et al., 1999; Jacob and Briese, 2003; Coombs et al., 2004; Smith, 2006).

2. Materials and methods

2.1. Insect population

The parental generation and descendants of a colony of *C. basicorne* held in the USDA-ARS quarantine laboratory in Albany, CA were used for these experiments. The colony was established from adults reared from naturally infested,

wild yellow starthistle plants that were collected at 15 sites near Kayseri, Sivas, Erzincan, Erzurum, and Malatya, Turkey between 28 May and 2 June 2001 (Smith and Drew, 2006). Emerged adults were identified by the author before using them in experiments. Identification of representative specimens was confirmed by B.A. Korotyaev, and vouchers were deposited at the USDA-ARS Systematic Entomology Laboratory in Beltsville, MD. Reproductive diapause was terminated by holding adults in the dark at 5 °C for at least three months (Smith and Drew, 2006). Experiments were conducted between March 2002 and April 2005.

2.2. Test plants

Test plant species were selected following Technical Advisory Group (TAG) guidelines, which emphasize evaluation of native and economically important species (USDA-APHIS, 1998; see discussion below). We generally used plants grown from seed that were two to four months old and in the rosette stage (except for species that do not form a rosette). However, because of scarcity of seed, many of the *Saussurea americana* Eaton plants were transplanted from the field. Cuttings of *Hemizonia minthornii* Jepson were used instead of potted plants. Seeds for test plants were obtained from commercial sources or from the wild with the assistance of cooperators (see Acknowledgements). A representative specimen of each species was grown to maturity to provide herbarium vouchers that are kept at the USDA-ARS Western Regional Research Center, Albany, CA. Identifications were confirmed by G.F. Hrusa (California Department of Food and Agriculture).

2.3. No-choice tests

Individual mated females that had completed reproductive diapause were held in a sealed container with a cut leaf of yellow starthistle, inserted in a water vial, until she oviposited (Fig. 1A). Each female was then placed in a clear plastic

tube (3.5 cm diameter × 11 cm long) mounted on an intact rosette leaf of a nontarget plant species for four to 5 days (Fig. 1B). Afterwards, we put each female back with a cut yellow starthistle leaf for two to three days to determine if she could still oviposit. If the female failed to oviposit on the post-trial yellow starthistle or died during the experiment, then the trial was considered invalid and the experiment was repeated. After removing the insect from the test plant, the exposed leaf was labeled, and we counted the number of adult feeding holes and eggs oviposited. After 10–21 days, which allowed time for eggs to hatch and larvae to tunnel down the petiole and into the root crown, the leaf was removed and examined under a microscope for signs of egg hatch and larval tunneling (see Smith and Drew, 2006). Six weeks after exposure to oviposition, each plant was enclosed in a fine mesh bag and held in a quarantine greenhouse until the insects could complete development (three months), then the plants were dissected to observe signs of insect damage and development. Any plants that deteriorated prematurely were dissected immediately. In general, we tested eight replicates per plant species in the tribe Cardueae and four in the more distantly related taxa. We doubled the number of replicates if there were any signs of larval development.

In general, no statistical tests were conducted on the results, because the purpose was to describe the risk and amount of damage or oviposition rather than to test hypotheses. However, χ^2 tests of independence were conducted to compare adult feeding damage and oviposition rates among the varieties of safflower tested to determine if any were more susceptible than the others.

3. Results and discussion

3.1. Test plants

Test plant species were selected following Technical Advisory Group (TAG) guidelines, which emphasize protection of native and economically important species

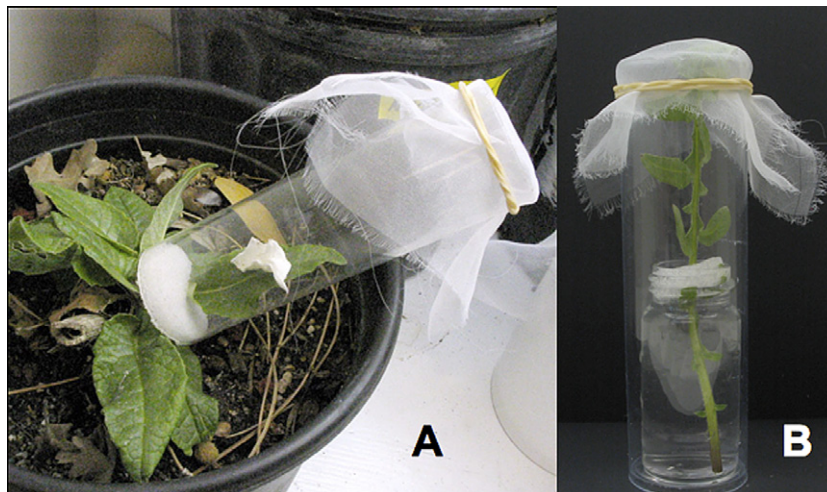


Fig. 1. Individual females were held in a sealed plastic cylinder on an intact leaf of a nontarget test plant for 5 days (A), then placed in a tube with a cut yellow starthistle leaf (B) to feed and oviposit before use in a subsequent test.

(USDA-APHIS, 1998; TAG, 2006). The guidelines generally follow the “centrifugal phylogenetic” approach outlined by Wapshere (1974), in which more species are tested in taxonomic ranks closely related to the target weed, and the number of test species decreases as relatedness to the target decreases. The validity of this approach is supported by the historical data that indicates that close relatives are most likely to suffer damage (Pemberton, 2000; Sheppard et al., 2005). Other factors that contributed to the choice of test species included: nativity in North America; ornamental or other economic value; whether the species is sympatric with the target’s present or potential range in North America; similarity of growth form, life history and secondary chemistry, if known; the presence of rare or protected species in the same genus; and availability of the species for testing. For rare or protected species that were proposed for testing, we often tested a close relative to avoid negatively impacting an already stressed species and/or because of unavailability of specimens. Species names are based on the PLANTS Database (USDA-NRCS, 2006) with the support of other regional flora, primarily Barkley (1986), Keil (1993) and Hitchcock and Cronquist (1998). Test plant species were selected following the higher taxonomy of Bremer (1994). A recent phylogenetic revision (Funk et al., 2005) has changed some taxonomic relationships; however the level of significance of the cladogram branches was not reported, so the relationships should be interpreted as the current, though possibly imperfect, state of knowledge. In particular, the subfamily Cichorioideae was reduced and Carduoideae was erected. The tribe Cardueae was previously placed in the subfamily Cichorioideae s.l.; however, the latter was determined to be a paraphyletic grade (Garcia-Jacas et al., 2002) and has since been redefined as a monophyletic group that does not include Cardueae (Funk et al., 2005). The tribe Mutisieae was removed from Cichorioideae s.s. and is now a basal, probably still paraphyletic, clade of the Asteraceae (Funk et al., 2005).

Yellow starthistle is in the family Asteraceae, subfamily Carduoideae, tribe Cardueae, and subtribe Centaureinae (Bremer, 1994; Funk et al., 2005). Both the subfamily Carduoideae and the tribe Cardueae appear to be monophyletic groups. The exact taxonomic relationships within the tribe Cardueae are not completely understood, but the subtribe Centaureinae segregates as a monophyletic group from the rest of the tribe, whereas the subtribe Carduinae is a paraphyletic grade (Bremer, 1994; Susanna et al., 1995; Garcia-Jacas et al., 2002). However, of the Carduinae genera of interest in this study, *Carduus*, *Cirsium*, *Cynara*, and *Silybum* appear to be in a monophyletic group, whereas *Onopordum* is positioned more basally (Funk et al., 2005). So, all these genera are more distantly related to *Centaurea* than are those in the Centaureinae. The monophyletic *Arctium*–*Cousinia*–*Saussurea*–*Jurinea* group within the Centaureinae is considered to be the closest related monophyletic group that includes *Centaurea*, *Acroptilon*, *Carthamus*, *Cnicus*, and probably *Crupina* (Garcia-Jacas et al., 2002; Funk et al., 2005, J.F. Gaskin, unpublished data).

The two subtribes Centaureinae and Carduinae have distinctly different secondary chemical compounds (Susanna et al., 1995), which are probably important in determining host plant specificity of specialist herbivores. Centaureinae produce acetylene aldehydes, chlorhydrins, and acetates, germacronolide-type sesquiterpenoids, highly methoxylated flavonoids (including flavanones), and fully methoxylated lignans (Wagner, 1997). In contrast, the Carduinae produce distinctive classes of acetylenes, including C₁₇ acetylenes and acetylene glycosides, guainolide-type sesquiterpenoids, monomethoxylated flavonoids, and simple cinnamic acids and their derivatives.

The genus *Centaurea* is very large, comprising 200–600 species, and its definition and extent is still being resolved (Klokov et al., 1963; Dostál, 1976; Susanna et al., 1995; Garcia-Jacas et al., 2000; Hellwig, 2004). Some groups within the genus *Centaurea* (e.g., *Centaurea sensu stricto*, *Cyanus*, *Jacea*, and *Psephellus* groups) appear to be as phylogenetically distinct as other well recognized genera (e.g., *Amberboa*, *Carduncellus*, *Carthamus*, *Cnicus*, *Crupina*, and *Serratula*) (Garcia-Jacas et al., 2001; J.F. Gaskin, unpublished data). Two North American species, *Centaurea americana* Nutt. and *Centaurea rothrockii* Greenm. have been assigned to the genus *Plectocephalus*, which has distinct pollen morphology and is thought to have diverged from the *Centaurea* clade during late Oligocene and Miocene (Wagenitz, 1955; Hellwig, 2004). This is much earlier than the divergence of *Cyanus* and the *Carthamus*/*Carduncellus* groups, which probably arose during the Pliocene–Pleistocene transition. The *Flora of North America* (Keil and Ochsmann, 2006) uses the treatments: *Centaurea benedicta* (= *Cn. benedictus*), *Plectocephalus americana* and *Plectocephalus rothrockii*, although the PLANTS website has not adopted these changes (USDA-NRCS, 2006). Although many of the phylogenetic relationships among the remaining species within the genus *Centaurea* are not precisely known, some groups have clearly emerged. The strongest grouping reflects differences in pollen structure (Wagenitz, 1955) and DNA nucleotide base sequences (Susanna et al., 1995; J.F. Gaskin, unpublished data). Yellow starthistle is in the *Jacea* group, which is monophyletic and includes many of the other weedy *Centaurea* species adventive to North America.

Plants of economic interest in the family Asteraceae distributed within the geographic range of yellow starthistle in North America include artichoke (*Cynara scolymus* L.), safflower (*Ca. tinctorius* L.) and sunflower (*Helianthus annuus* L.). Because a population of *C. basicorne* from Italy has been reported to damage and develop in safflower (Clement et al., 1989), and this is an important crop in California and other western states, we evaluated nine varieties. Bachelor’s button (*Ce. cyanus*) is an introduced ornamental in North America, but is also considered an invasive weed in some areas of the western US (Lorenzi and Jeffery, 1987; Taylor, 1990), and is a common weed in wheat fields in eastern Europe (Voronov, 1977; Kapeluszný and Pawlowski, 1978; Snarska, 2004). The native North American plants

most closely related to yellow starthistle include: *Ce. americana* and *Ce. rothrockii* (discussed above), *S. americana*, and the many species of *Cirsium*. Several *Cirsium* species are becoming rare, and six species or varieties are federally listed as endangered (E) or threatened (T): *Cirsium fontinale* (Greene) Jepson var. *fontinale* (E), *Ci. fontinale* (Greene) Jepson var. *obispoense* J.T. Howell (E), *Ci. hydrophilum* (Greene) Jepson var. *hydrophilum* (E), *Cirsium loncholepis* Petrak (E), *Cirsium pitcheri* (Torr. ex Eat.) Torr. and Gray (T), and *Cirsium vinaceum* Woot. and Standl. (T) (USFWS, 2005). A total of 20 native species, including *Ci. fontinale* var. *fontinale* and *Ci. fontinale* var. *obispoense*, occur in California. Eleven California species are considered rare (Tibor, 2001), and *Cirsium ciliolatum* (Henderson) J.T. Howell and *Cirsium rhotophilum* Blake are listed by the state of California as endangered and threatened, respectively.

3.2. No-choice tests

We tested a total of 51 species of nontarget host plants from the Asteraceae family, including 25 native species and 4 economic species (Table 1). This includes species from all five genera in the subfamily Carduoideae, all three tribes in the subfamily Cichorioideae s.s., and all eight tribes in the subfamily Asteroideae that contain native North American species or economic species. In no-choice oviposition tests, *C. basicorne* females oviposited at least once on 94% of the plant species in the subtribe Centaureinae, including *Ca. tinctorius* (safflower) and the native species *Ce. americana* and *Ce. rothrockii*. At least one egg was deposited on 62% of the plant species in other subtribes of the Cardueae, and most frequently on *S. americana* and *Ci. loncholepis*. Eggs were observed on only three plants outside the tribe Cardueae: one egg on one plant of *Liatris punctata* Hook., two eggs on one plant of *Gazania rigens* (L.) Gaertn., and six eggs on one plant of *Pluchea odorata* (L.) Cass. None of the eggs on the first two plants hatched. Two eggs on *P. odorata* hatched, but larvae died when they reached the end of the petiole. These results indicate no risk of significant larval damage to plants outside the tribe Cardueae.

The highest rates of larval survival and development were observed on *Ce. solstitialis* (yellow starthistle) and *Ce. cyanus* (bachelor's button), and there was development on *Centaureinae melitensis* L. (Napa starthistle, tocalote), *Centaureinae montana* L., *Centaureinae nigrescens* Willd. (= *Centaureinae x pratensis*, meadow knapweed), *Centaureinae sulphurea* Willd., *Cn. benedictus* L. (blessed thistle), *Ca. tinctorius* (safflower), and *Crupina vulgaris* Cass. (common crupina). There was no larval development on any *Cirsium* species tested, which is consistent with Clement et al.'s (1989) results for *Cirsium douglasii* DC. and *Cirsium campylon* H. Sharsm. There was no larval development in any threatened or endangered species, nor their surrogates that we tested. Regarding the two North American native *Centaurea* species, no development was observed in 18 trials of *Ce. rothrockii* or 21 trials of *Ce. americana*. The roots

of two *S. americana* plants were damaged, but the damage was not consistent with that usually caused by *C. basicorne*. These plants had been collected from the field shortly before being tested, so it is likely that the damage was caused by other species of insect that had attacked the plant in the field. Other *S. americana* plants that were not infested by *C. basicorne* had similar damage. These results indicate that there is zero to very low risk that *C. basicorne* will damage any North American native plant species.

Our results generally corroborated those of Clement et al. (1989), indicating no larval development on any *Cirsium* species tested, *Cy. scolymus* (artichoke) or *Ce. calcitrapa* L. However, there were some discrepancies. Clement et al. (1989) reported larval development on *Car. pycnocephalus* L. in 11% of trials, in which four neonate larvae were transferred to each replicate plant, whereas we observed no development on *Car. pycnocephalus*, despite oviposition of ten eggs on one out of ten plants tested. The seven eggs oviposited in the leaf blade either did not hatch or the larvae failed to reach the midrib. Of the three eggs oviposited in the midrib, two larvae tunneled down the petiole, but no damage to the stem was observed. Apparently placing neonate larvae in a hole in the central rosette meristem is more conducive to larval development. On the other hand, we observed development of some larvae on *Cn. benedictus*, whereas Clement et al. (1989) observed no development on the three plants tested. Although we did not test *G. tomentosus* Moench, absence of development in any Cardueae outside the subtribe Centaureinae, suggests that we would not have observed development in this plant; however, Clement et al. (1989) reported development in 20% of their larval transfer trials. The differences between our results and those of Clement et al. (1989) indicate that estimates of host plant suitability that are based on transfer of neonate larvae to a potentially susceptible part of the plant (into a hole in the central meristem) can differ from those based on naturally oviposited eggs. The reason is probably because eggs and emerging larvae oviposited in the leaf blade and midrib face different plant defenses than larvae artificially placed in the central meristematic tissue. Thus, when designing larval transfer experiments, it is important to place larvae in as natural a location as possible to improve the validity of extrapolating the results to predict what would occur under natural conditions. Larval transfer is less preferable for insects that oviposit into plant tissue than for those that oviposit externally (Sheppard, 1999).

Development of larvae in safflower and bachelor's button may not be normal for *C. basicorne* because these plants do not form a rosette. Thus, when young larvae tunnel down a leaf on either of these plants, they cannot reach the root crown. The stem of these plants has a pithy center, and larvae only feed in the woody outer portion of the stem. The relatively thin cortex provides a limited space for the insect, and as the plant continues to grow, it sometimes crushes the pupae. Nevertheless, there was high infestation and survivorship to the pupal stage on bachelor's button and safflower in no-choice experiments, so both these

Table 1
Oviposition and immature development of *C. basicorne* on test plants in no-choice conditions (one female per plant, held in tube on a leaf for 5 days on nontarget species or for 2–3 days on yellow starthistle)

Plant species	No. of trials oviposition	Adult feeding holes/day (\pm SE)	Eggs/day (\pm SE)	No. of trials development ^a	Percentage of trials			
					Adult feeding holes present	Eggs present	Internal plant damage present	Adults or pupae present
Subfamily Cardioideae								
Tribe Cardueae								
Subtribe Centaureinae								
<i>Acroptilon repens</i> (L.) DC., Russian knapweed ^d	10	4.09 \pm 0.82	0.16 \pm 0.06	9	90	40	0	0
<i>Carthamus tinctorius</i> L., safflower	100	10.66 \pm 0.82	0.47 \pm 0.05	76	57	61	42	38
<i>Centaurea americana</i> Nutt., American basketflower ^b	23	6.04 \pm 1.42	0.22 \pm 0.08	21	91	39	0	0
<i>Centaurea calcitrapa</i> L., purple starthistle ^d	8	9.59 \pm 2.20	0.25 \pm 0.08	8	100	75	0	0
<i>Centaurea cineraria</i> L., dusty miller ^c	10	0.51 \pm 0.13	0.00 \pm 0.00	10	80	0	0	0
<i>Centaurea cyanus</i> L., cornflower, bachelor's button ^{c,d}	8	22.66 \pm 3.30	1.73 \pm 0.29	8	100	100	100	100
<i>Centaurea dealbata</i> Willd., whitewash cornflower ^d	11	10.71 \pm 2.54	0.85 \pm 0.30	13	100	82	9	0
<i>Centaurea diffusa</i> Lam., diffuse knapweed ^d	8	24.28 \pm 5.22	1.50 \pm 0.43	7	100	88	0	0
<i>Centaurea stoebe</i> L. (= <i>maculosa</i>), spotted knapweed ^d	8	8.84 \pm 1.78	0.56 \pm 0.13	6	100	75	0	0
<i>Centaurea melitensis</i> L., Napa starthistle, tocalote ^d	20	11.25 \pm 2.01	0.60 \pm 0.12	17	95	75	44	44
<i>Centaurea montana</i> L., perennial cornflower ^c	17	4.31 \pm 2.04	0.25 \pm 0.15	15	53	35	13	7
<i>Centaurea nigrescens</i> Willd. (= <i>C. x pratensis</i>), meadow knapweed ^d	14	10.24 \pm 2.25	0.74 \pm 0.21	15	93	64	42	10
<i>Centaurea rothrockii</i> Greenm., Rothrock's basketflower ^b	18	12.83 \pm 1.91	0.30 \pm 0.09	15	100	56	0	0
<i>Centaurea solstitialis</i> L., yellow starthistle ^d	349	16.58 \pm 0.46	1.54 \pm 0.05	32	100	99	89	88
<i>Centaurea sulphurea</i> Willd., Sicilian starthistle ^d	13	5.18 \pm 1.13	0.34 \pm 0.13	12	77	54	10	10
<i>Centaurea virgata</i> Lam. ssp. <i>squarrosa</i> (Willd.) Gugler, squarrose knapweed ^d	16	7.91 \pm 1.11	0.51 \pm 0.12	10	100	81	0	0
<i>Cnicus benedictus</i> L., blessed thistle ^d	8	5.58 \pm 1.14	0.50 \pm 0.17	9	100	88	50	38
<i>Crupina vulgaris</i> Cass., common crupina ^d	9	3.51 \pm 1.12	0.24 \pm 0.13	11	89	44	22	22
Subtribe Carduinae								
<i>Carduus pycnocephalus</i> L., Italian thistle ^d	10	1.74 \pm 1.63	0.20 \pm 0.20	9	40	10	0	0
<i>Cirsium brevistylum</i> Cronq., Indian thistle ^b	7	0.00 \pm 0.00	0.11 \pm 0.07	7	0	29	0	0
<i>Cirsium ciliolatum</i> (Henderson) Howell, Ashland thistle ^b	9	0.13 \pm 0.09	0.00 \pm 0.00	9	22	0	0	0
<i>Cirsium cymosum</i> (Greene) Jepson, peregrine thistle ^b	11	0.10 \pm 0.06	0.00 \pm 0.00	8	18	0	0	0
<i>Cirsium fontinale</i> Greene var. <i>fontinale</i> , fountain thistle ^b	15	0.06 \pm 0.03	0.00 \pm 0.00	15	27	0	0	0
<i>Cirsium hydrophilum</i> (Greene) Jepson var. <i>vaseyi</i> (Gray) Howell, Mount Tamalpais thistle ^b	16	0.70 \pm 0.39	0.07 \pm 0.05	16	56	13	0	0
<i>Cirsium loncholepis</i> Petrak, la graciosa thistle ^b	9	4.04 \pm 1.92	0.36 \pm 0.24	9	78	56	0	0
<i>Cirsium occidentale</i> (Nutt.) Jepson var. <i>venustum</i> (Greene) Jepson, venus thistle ^b	16	1.40 \pm 1.09	0.03 \pm 0.03	15	44	6	0	0
<i>Cirsium vinaceum</i> Woot. and Standl., Sacramento Mountain thistle ^b	7	0.00 \pm 0.00	0.00 \pm 0.00	7	0	0	0	0
<i>Cirsium vulgare</i> (Savi) Ten., bull thistle ^d	8	0.44 \pm 0.44	0.06 \pm 0.06	8	13	13	0	0
<i>Cynara scolymus</i> L., artichoke	11	1.18 \pm 0.64	0.00 \pm 0.00	11	55	0	0	0
<i>Onopordum acanthium</i> L., Scotch thistle ^d	8	0.75 \pm 0.30	0.06 \pm 0.04	8	88	25	0	0
<i>Saussurea americana</i> Eaton, American sawwort ^b	41	4.04 \pm 0.91	0.35 \pm 0.07	23	73	51	0 ^c	0 ^c
<i>Silybum marianum</i> (L.) Gaertn., milk thistle ^d	8	0.19 \pm 0.10	0.00 \pm 0.00	8	38	0	0	0
Subtribe Carlininae								
<i>Xeranthemum cylindraceum</i> Sibth. and Sm., lilac stars ^c	4	3.35 \pm 2.89	0.25 \pm 0.25	4	75	25	0	0
Subtribe Echinopsidinae								
<i>Echinops exaltatus</i> Schrad., tall globethistle ^c	4	1.85 \pm 0.88	0.05 \pm 0.05	4	100	25	0	0

(continued on next page)

Table 1 (continued)

Plant species	No. of trials oviposition	Adult feeding holes/day (\pm SE)	Eggs/day (\pm SE)	No. of trials development ^a	Percentage of trials			
					Adult feeding holes present	Eggs present	Internal plant damage present	Adults or pupae present
Subfamily Cichorioideae s.s.								
Tribe Arctoteae								
<i>Gazania rigens</i> (L.) Gaertn., treasure-flower ^c	10	2.20 \pm 1.12	0.04 \pm 0.04	10	40	10	0	0
Tribe Lactuceae								
<i>Agoseris grandiflora</i> (Nutt.) Greene, bigflower agoseris ^b	8	0.00 \pm 0.00	0.00 \pm 0.00	8	0	0	0	0
<i>Stephanomeria cichoriacea</i> Gray, chicoryleaf wirelettuce ^b	4	0.00 \pm 0.00	0.00 \pm 0.00	2	0	0	0	0
<i>Lactuca sativa</i> L., garden lettuce	8	0.00 \pm 0.00	0.00 \pm 0.00	8	0	0	0	0
Tribe Vernoniaeae								
<i>Stokesia laevis</i> (Hill) Greene, Stokes' aster ^b	4	0.00 \pm 0.00	0.00 \pm 0.00	4	0	0	0	0
Subfamily Asteroideae								
Tribe Anthemideae								
<i>Artemisia californica</i> Less., coastal sagebrush ^b	6	0.33 \pm 0.33	0.00 \pm 0.00	6	17	0	0	0
Tribe Astereae								
<i>Symphyotrichum</i> (= <i>Aster</i>) <i>chilense</i> (Nees) Nesom var. <i>chilense</i> , Pacific aster ^b	5	0.00 \pm 0.00	0.00 \pm 0.00	5	0	0	0	0
Tribe Eupatorieae								
<i>Brickellia californica</i> (Torr. and Gray) Gray, California brickellbush ^b	4	0.35 \pm 0.17	0.00 \pm 0.00	4	75	0	0	0
<i>Liatris punctata</i> Hook., dotted blazing star ^b	5	0.72 \pm 0.40	0.04 \pm 0.04	4	60	20	0	0
Tribe Gnaphalieae								
<i>Pseudognaphalium</i> (= <i>Gnaphalium</i>) <i>californicum</i> (DC.) A. Anderb., ladies' tobacco ^b	6	0.00 \pm 0.00	0.00 \pm 0.00	5	0	0	0	0
Tribe Helenieae								
<i>Eriophyllum staechadifolium</i> Lag., seaside woolly sunflower ^b	7	0.37 \pm 0.20	0.00 \pm 0.00	7	43	0	0	0
<i>Hemizonia minthornii</i> Jepson, Santa Susana tarweed ^b	4	0.00 \pm 0.00	0.00 \pm 0.00	4	0	0	0	0
Tribe Heliantheae								
<i>Echinacea purpurea</i> (L.) Moench, eastern purple coneflower ^b	6	0.17 \pm 0.17	0.00 \pm 0.00	6	17	0	0	0
<i>Helianthus annuus</i> L., common sunflower ^b	4	0.00 \pm 0.00	0.00 \pm 0.00	4	0	0	0	0
Tribe Inuleae (formerly in Plucheeae)								
<i>Pluchea odorata</i> (L.) Cass., salt marsh fleabane ^b	6	0.13 \pm 0.10	0.20 \pm 0.20	6	33	17	0	0
Tribe Senecioneae								
<i>Senecio cineraria</i> DC., silver ragwort ^c	8	0.00 \pm 0.00	0.00 \pm 0.00	6	0	0	0	0
<i>Senecio vulgaris</i> L., old-man-in-the-Spring ^d	4	0.35 \pm 0.35	0.00 \pm 0.00	4	25	0	0	0
Grade 1 (formerly in Cichorioideae)								
Tribe Mutisieae s.s.								
<i>Trixis californica</i> Kellogg, American threefold ^b	5	0.12 \pm 0.08	0.00 \pm 0.00	5	40	0	0	0

All plants are in the family Asteraceae. Higher taxonomy is based on Bremer (1994) and Funk et al. (2005).

^a Number of trials for insect development was less than the number of oviposition trials when plants were discarded because of deterioration caused by other factors.

^b Native to North America.

^c Alien ornamental.

^d Alien weed.

^e Two *S. americana* plants were damaged, but the damage was not consistent with *C. basicorne*. Other plants not infested by *C. basicorne* had similar damage, which was presumably caused by infestation by other insects in the field before entry to quarantine laboratory.

plants require additional choice testing in the laboratory and field to determine risk of infestation under more natural conditions (Smith et al., 2006).

The host plants suitable for *C. basicorne* development correspond to a clearly defined phylogenetic group. Our results indicate that *C. basicorne* is able to develop on a small number of plants within a monophyletic “derived clade” within the subtribe Centaureinae that includes the Jacea, Cyanus and Carthamus groups (Fig. 2). Delineation of this clade is based on nucleotide sequences of ITS nuclear ribosomal DNA and agrees with other morphological characters (Garcia-Jacas et al., 2001); however, the relationships within it are not fully certain because of low bootstrap values (J.F. Gaskin, unpublished data). The clade includes the suitable species: *Ce. solstitialis*, *Ce. melitensis*, *Ce. nigrescens*, *Ce. sulphurea* and *Cn. benedictus* in the Jacea group, *Ce. cyanus* and *Ce. montana* in the Cyanus

group, and *Ca. tinctorius* in the Carthamus group (Garcia-Jacas et al., 2001; J.F. Gaskin, unpublished data). Within this clade, there are no native North American plants, and the only plants of economic interest are *Ca. tinctorius*, a significant crop, and *Ce. cyanus*, an ornamental. The two native North American species, *Ce. americana* and *Ce. rothrockii*, which were not suitable hosts, are in a different clade and have recently been assigned to the genus *Plectocephalus* (Susanna et al., 1995; Hellwig, 2004). *Cr. vulgaris* may be an outlier because it appears to be more distantly related to *Ce. solstitialis* than other groups containing unsuitable plants: *Acroptilon*, *Plectocephalus* and *Psephellus*. Some species within the “derived clade” were not suitable for *C. basicorne* development: *Ce. calcitrapa*, *Ce. cineraria*, *Centaurea diffusa*, and *Centaurea stoebe* (often called *Centaurea maculosa* in North America (Ochsmann, 2001)). So, although the “derived clade” includes almost all

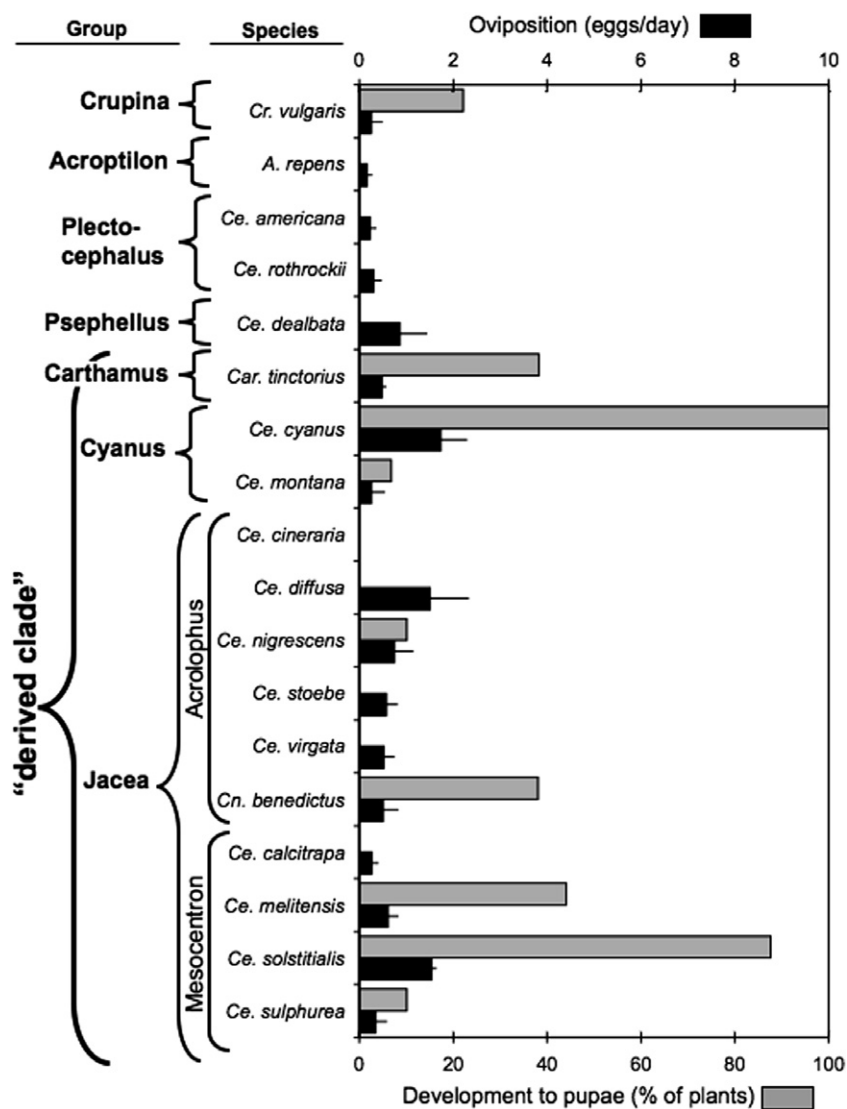


Fig. 2. Suitability of plants within the subtribe Centaureinae to oviposition and larval development by *C. basicorne*. Classification of the species into “groups” is based on plant morphology, pollen type and DNA sequences of introns (Garcia-Jacas et al., 2001; J.F. Gaskin, unpublished data). Oviposition is mean eggs per female per day of exposure to the plant ($\pm 95\%$ CI), and development is percentage of oviposition trials in which at least one insect developed to at least the pupal stage.

species that are suitable, not all species within the clade are suitable. These results confirm that although phylogenetic relationships explain a high degree of host plant specificity, they do not explain all of it. The exceptions presumably are caused by evolutionary divergence of critical characters (e.g., allelochemicals and plant morphology) in close relatives that cause them to be less suitable, and evolutionary convergence in more distant relatives that make them more suitable. Thus, selection of plant species to test should not rely only on phylogeny, as some have proposed (Briese, 2005, 2006), lest we overlook a more distant relative that may be suitable because of similarity of critical characters (e.g., Wheeler, 2005; Haines et al., 2004).

All the reported larval host plants from field collections: *Ce. solstitialis*, *Ce. cyanus*, and *Cn. benedictus* (Alonso-Zarazaga, 1990a; Wanat, 1994; Campobasso et al., 1999; J. Balciunas, unpublished data) were suitable in the laboratory experiments. However, other species that were suitable in the laboratory, such as *Ca. tinctorius*, *Ce. melitensis*, *Ce. sulphurea*, *Ce. nigrescens* and *Ce. montana*, have not been reported as a host in the field. This agrees with current theory that the physiological range, delineated in no-choice laboratory experiments, is broader than the ecological range realized in the field (Briese, 2005; Sheppard et al., 2005). Field experiments conducted in Turkey indicated that *Ca. tinctorius* was not attacked by *C. basicorne*, despite natural infestation of 48–98% of adjacent *Ce. solstitialis* plants (Smith et al., 2006). Thus, *Ca. tinctorius* is not likely to be attacked in the field. Similar experiments have not been done for *Ce. cyanus*, so risk to this species in the field is not well known.

Intensity of adult feeding on leaves was highly correlated to the number of eggs oviposited in test plants ($R^2=0.88$, $P<0.0001$; Fig. 3), probably because adult feeding is necessary for egg development. Adult feeding damage was highest on *Ce. solstitialis* (yellow starthistle), *Ce. cyanus* (bachelor's button) and *Ce. diffusa* Lam. (diffuse knapweed) (Fig. 3 and Table 1). Feeding rate on *Ce. solstitialis* may have been underestimated because (1) exposure was usually for 3 days on cut leaves versus for 5 days on intact plants

for nontarget species, and (2) it included the “positive control” trials that followed exposure to nontarget plants, many of which may have negatively affected the insects. There was moderate acceptance of nine other species of *Centaurea*, *Ca. tinctorius* (safflower) and *Cn. benedictus*. Low adult feeding occurred on the other Centaureinae, about half the other Cardueae, and one other species of Cichorioideae (*G. rigens*). There was, at most, only trace feeding on test plants in the subfamily Asteroideae. These results suggest that under extreme conditions *C. basicorne* adults may feed on other species of plants, particularly in the subtribe Centaureinae. Risk of adult feeding damage is generally limited to plants within the tribe Cardueae. Each adult feeding hole is about 1-mm², and they were smaller on most nontarget species. Therefore, adult feeding is not expected to cause any noticeable damage to nontarget species except possibly to *Ce. cyanus* and *Ce. diffusa*.

3.3. Safflower varieties

All nine safflower varieties were susceptible to adult feeding and oviposition under no-choice conditions (Table 2). Adult feeding (7.4–16.2 holes/days) and oviposition (0.3–0.8 eggs/days) rates tended to be lower than on yellow starthistle (16.6 and 1.5, respectively) but are clearly a concern regarding susceptibility of this plant. Larval damage and development to at least the pupal stage occurred in 30–50% of plants tested, depending on variety. There were no significant differences among varieties (χ^2 , $df=9$, $P>0.5$), even when varieties with less than ten replicates were excluded from analysis. These results confirm those of Clement et al. (1989) indicating that *C. basicorne* is physiologically able to feed, oviposit and develop on safflower under no-choice laboratory conditions.

3.4. Relationship of host range to *Ceratapion* phylogeny

The genus *Ceratapion* contains 55 species and subspecies, divided among five subgenera: *Acanephodous*, *Ceratapion*, *Clementiellus*, *Echinostroma*, and *Angustapion* (Wanat, 1994). Most speciation occurred in Pliocene–Miocene, with the last events in the Pleistocene Glacial Period (including vicariance of sibling species). Species are distributed almost throughout the entire Palearctic. In general, larvae and adults of species in the genus *Ceratapion* feed only on Asteraceae in the tribe Cardueae (=“Cynareae”) (Alonso-Zarazaga, 1990b). Genera recorded as food plants are: *Arctium*, *Carduus*, *Centaurea*, *Cirsium*, *Cynara*, *Echinops*, *Galactites*, *Onopordum*, *Silybum*, and *Xeranthemum*. Hypotheses of the phylogenetic relationships among species of *Ceratapion* have been proposed by Alonso-Zarazaga (1990a) and Wanat (1994), based on morphological characters (Fig. 4). Species in the subgenus *Echinostroma*, which includes *C. basicorne*, are associated with plants in the genera *Arctium*, *Carlina*, *Carthamus*, *Centaurea* and *Silybum* (Alonso-Zarazaga, 1990a; Wanat, 1994). Larval host plants of *Ceratapion curtii* (Wagner), the closest relative of

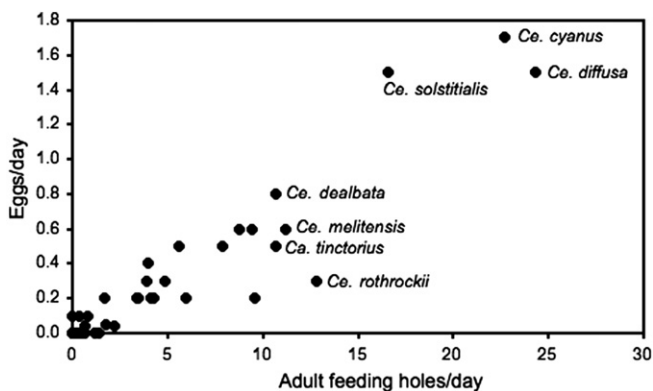


Fig. 3. Relationship of mean number of adult feeding holes (ca. 1-mm²) to mean number of eggs on the different test plant species under no-choice conditions. Each point represents a different test plant species (YST = *Centaurea solstitialis*, safflower = *Ca. tinctorius*).

Table 2

Physiological suitability of different varieties of safflower to oviposition and development of *Ceratapion basicorne* in a no-choice oviposition experiment

Safflower variety ^a	No. of trials oviposition	Adult feeding holes/day	Eggs/day	No. of trials development	Percentage of trials			
					Adult feeding holes present	Eggs present	Internal plant damage present	Adults or pupae present
CW-88-OL ^b	23	10.7	0.6	10	91	61	50	50
CW-1221	16	16.2	0.4	16	44	63	31	31
CW-4440	16	9.1	0.5	15	38	63	40	40
Gila ^c	10	7.4	0.4	—	80	80	—	—
Hartman ^d	7	—	0.3	7	—	43	43	43
S-345-OL	6	14.8	0.8	6	100	67	50	33
S-400 ^d	2	—	0.3	2	—	100	50	0
S-518-OL	10	8.4	0.4	10	90	50	40	30
S-541 ^d	10	—	0.4	10	—	50	50	50

^a Seed producers: CW, CalWest; S, SeedTec; OL, high oleic oil content.

^b Cut leaves in vials were used in 12 trials of CW-88, for which no development data are available.

^c No development data are available because plants died during malfunction of cooling system.

^d Adult feeding holes were not counted.

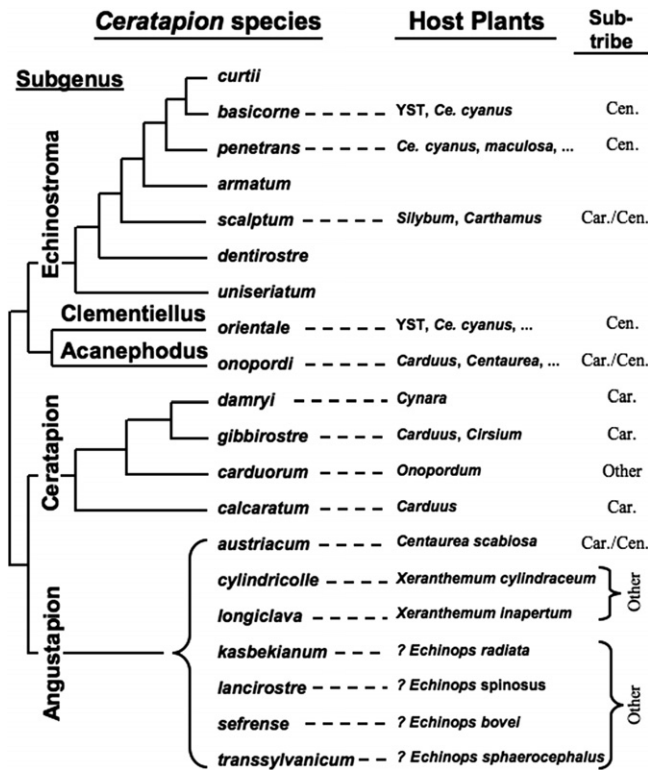


Fig. 4. Phylogenetic relationships among species and subgenera in the genus *Ceratapion* are after Alonso-Zarazaga (1990a) and Wanat (1994). Larval host records are from both authors and Balciunas (unpublished data). Hosts of *C. curtii*, *C. armatum*, *C. dentiostre* and *C. uniseriatum* are unknown. All host plants are in the tribe Cardueae, and subtribe assignments are from Funk et al. (2005). (YST, *Ce. solstitialis*; "...", other Cardueae species; "?", uncertain host plant).

C. basicorne, are unknown. Reported hosts of the next closest relative, *Ceratapion penetrans* (Germar), which has three subspecies, are: *Centaurea rhenana* [= *stoebe*, = *maculosa*, = *paniculata*], *Centaurea jacea*, *Ce. cyanus*, *Ce. diffusa*, *Centaurea nigra*, *Centaurea scabiosa*, and *Ce. solstitialis* (Alonso-Zarazaga, 1990a; Wanat, 1994; Balciunas unpublished data). Older reports of *Arctium lappa* and *Carlina*

vulgaris (Ehret, 1983) as hosts for this species may be mistaken. *Ceratapion scalptum* (Mulsant and Rey), which is also in the same subgenus, has one subspecies that develops on *Carthamus* and another on *Silybum*. Thus, each of the species or subspecies in the subgenus *Echinostroma* appear to develop only on plants within a genus. The historical restriction of evolutionary radiation of species within the genus *Ceratapion* to host plants within the Cardueae, and of species within the subgenus *Echinostroma* to a few genera within the Cardueae, suggests that *C. basicorne* is not likely to ever adapt to host plants outside this tribe. Interestingly, the two species that attack crops, *Cer. scalptum* (Mulsant and Rey) on safflower and *Ceratapion damryi* (Desbrochers) on artichoke (*Cy. scolymus*), are very specific and have never been reported to develop on *Ce. solstitialis* or *Ce. cyanus*. So, conversely, it is not surprising that *C. basicorne*, which is specific to the latter species, does not develop on either of the former species, at least under field conditions (Smith et al., 2006).

The current theories on the phylogeny of species within the tribe Cardueae (Bremer, 1994; Susanna et al., 1995; Garcia-Jacas et al., 2002; Funk et al., 2005) and the genus *Ceratapion* (Alonso-Zarazaga, 1990a; Wanat, 1994) suggest that there is not an overall one-to-one pattern of *Ceratapion* species coevolving with Cardueae species (Fig. 4). For example, *Ceratapion onopordi* appears to be relatively polyphagous, being reported from hosts in six genera from both the Centaureinae and Carduinae (Alonso-Zarazaga, 1990a; Wanat, 1994). The two *Cer. scalptum* subspecies attack different genera in different subtribes (*Carthamus* spp. [Centaureinae] and *Silybum marianum* [Carduinae]), suggesting taxonomically how far a species in this group can jump when adopting a new host. However, most of the species in the subgenera *Echinostroma*, *Clementiellus* and *Acanephodus*, for which host plants are known, use hosts in the Centaureinae (especially *Centaurea* and *Carthamus* species); however, *Silybum* is in the Carduinae. Species in the subgenus *Ceratapion* are associated with Carduinae (*Carduus*, *Cirsium*, *Cynara*, and *Onopordum*), and those in

the subgenus *Angustapion* are generally associated with more basal taxa of the Cardueae (*Echinops*, *Xeranthemum*), but one species is associated with *Ce. scabiosa*.

Although all *Ceratapion* species are restricted to plants in the tribe Cardueae, they vary in their degree of host plant specificity. Because all species within the subgenus *Echinostroma* are only known to develop on plants in the Centaureinae and Carduinae, it appears that *C. basicorne* and its recent ancestors have been specializing on these host plants for a long time. This suggests that host plant specificity is deeply ingrained and that the insect is not likely to drastically change its host range. Thus, it appears that if *C. basicorne* were to adopt a new host after being released in North America, it would most likely be a close relative of *Ce. solstitialis* and *Ce. cyanus*. The closest native species are *Ce. americana* and *Ce. rothrockii*, but they are so distantly related as to be considered in a different genus (*Plectocephalus*), and they were not able to sustain larval development in our no-choice experiments. Therefore, it seems improbable that the insect could adapt to developing on any native North American species in the foreseeable future.

4. Conclusions

The no-choice results indicate that no plant species outside the subtribe Centaureinae are at risk of significant larval damage. Although adult feeding on foliage and oviposition occurred on many nontarget plants under no-choice conditions, they were at much lower rates than on *Ce. solstitialis*, *Ce. cyanus* and *Ce. diffusa*. Under choice conditions, the nontarget attack rates would be expected to be lower. Because *C. basicorne* is synovigenic, and must feed to continue producing eggs, absence of its preferred host plant would probably reduce egg production and consequently the risk of oviposition on nontarget plants. The oviposition rates that we observed on nontarget plants are probably elevated because of feeding on *Ce. solstitialis* prior to exposure to each nontarget test plant. Nontarget plants that supported oviposition and larval development warrant further evaluation under choice conditions to further assess the degree to which they are at risk. These include the two cultivated species: *Ca. tinctorius* (safflower) and *Ce. cyanus* (bachelor's button), and the native North American species: *Ce. americana*, *Ce. rothrockii*, *Ci. loncholepis* and *S. americana*. Larval damage to *Acroptilon repens*, *Cn. benedictus*, *Cr. vulgaris* and the other *Centaurea* species is acceptable because are all alien noxious weeds in North America.

Successful development of an insect on a crop in laboratory experiments is usually sufficient to discourage further evaluation of it as a biological control agent. However, the absence of field records of *C. basicorne* developing on safflower, despite records of its close relative, *Cer. scalptum*, on this plant, suggest that risk to safflower under field conditions may be insignificant. Only by conducting further choice experiments under laboratory or field conditions can we improve our estimation of this risk. Field trials that were

conducted at sites with natural populations of *C. basicorne* in Turkey showed no larval development on safflower by *C. basicorne* (Smith et al., 2006). However, three other insects did develop on safflower: *Cer. scalptum*, *Ceratapion orientale*, and *Ceratapion onopordi*. Therefore, if *C. basicorne* is approved for release, it will be important to correctly identify all specimens to prevent accidental introduction of any of these species. Taxonomic keys with detailed illustrations have been developed to assist proper identification of *C. basicorne* (J.K. Balciunas and B.A. Korotyaev, unpublished manuscript).

Centaurea cyanus (bachelor's button) is at risk of adult feeding, oviposition and larval damage. This plant has previously been reported as a host of *C. basicorne* in its native range in Eurasia, but the frequency of such attack has not been studied. The plant is both an ornamental and an invasive weed in parts of North America. The developing larva causes a small swelling of the stem at the base of the petiole down which it tunneled. Although this may cause cosmetic damage to the ornamental, it is not known how frequently this may occur, especially when the plant is grown as an intensively managed monoculture. Because this plant is an invasive weed in some regions of the western US, damage to it in rangeland habitats would be beneficial.

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**Decision and Finding of No Significant Impact
for
Field Release of *Ceratapion basicorne* (Coleoptera: Apionidae), a Weevil for Biological
Control of Yellow Starthistle (*Centaurea solstitialis*), in the Continental United States
August 2019**

The U.S. Department of Agriculture (USDA), Animal and Plant Health Inspection Service (APHIS) is proposing to issue permits for release of a weevil, *Ceratapion basicorne* (Illiger) (Coleoptera: Apionidae), in the continental United States. This agent would be used for the biological control of yellow starthistle, *Centaurea solstitialis* L. (Asteraceae). Before permits are issued for release of *C. basicorne*, APHIS must analyze the potential impacts of the release of this organism into the continental United States. APHIS has prepared an environmental assessment (EA) that analyzes the potential environmental consequences of this action. The EA is available from:

U.S. Department of Agriculture
Animal and Plant Health Inspection Service
Plant Protection and Quarantine
Pests, Pathogens, and Biocontrol Permits
4700 River Road, Unit 133
Riverdale, MD 20737

http://www.aphis.usda.gov/plant_health/ea/index.shtml

The EA analyzed the following two alternatives in response to the need to control yellow starthistle and contain infestations: (1) no action, and (2) issue permits for the release of *C. basicorne* for biological control of yellow starthistle. A third alternative, to issue permits with special provisions or requirements concerning release procedures or mitigating measures, was considered. However, this alternative was dismissed because no issues were raised that indicated that special provisions or requirements were necessary. The No Action alternative, as described in the EA, would likely result in the continued use at the current level of chemical, mechanical, cultural, and biological control methods for the management of yellow starthistle. These control methods described are not alternatives for decisions to be made by APHIS, but are presently being used to control yellow starthistle in the United States and may continue regardless of permit issuance for field release of *C. basicorne*. Notice of the EA was made available in the Federal Register on March 11, 2009 for a 30-day public comment period. One comment was received on the EA from the Tribal Historic Preservation Officer of the Bois Forte Band of Ojibwe in Minnesota. The commenter indicated a general disagreement with biological control but did not raise any specific issues regarding the release of the organism.

I have decided to authorize APHIS to issue permits for the environmental release of *C. basicorne*. The reasons for my decision are:

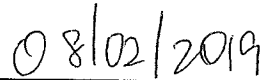
- This biological control agent is sufficiently host specific and poses little, if any, threat to the biological resources of the continental United States.

- The release not likely to adversely affect federally listed threatened and endangered species or their habitats in the continental United States. APHIS received a letter from the U.S. Fish and Wildlife Service, concurring with this determination.
- *C. basicorne* poses no threat to the health of humans or wild or domestic animals.
- No negative cumulative impacts are expected from release of *C. basicorne*.
- There are no disproportionate adverse effects to minorities, low-income populations, or children in accordance with Executive Order 12898 “Federal Actions to Address Environmental Justice in Minority Populations and Low-income Populations” and Executive Order 13045, “Protection of Children from Environmental Health Risks and Safety Risks.”
- While there is not total assurance that the release of *C. basicorne* into the environment will be reversible, there is no evidence that this organism will cause any adverse environmental effects.

I have determined that there would be no significant impact to the human environment from the implementation of the preferred alternative and, therefore, no Environmental Impact Statement needs to be prepared.



Steven Crook
Director of Permitting and Coordination Compliance
Plant Protection and Quarantine
Animal and Plant Health Inspection Service
U.S. Department of Agriculture



Date