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Nature of Petition:

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

Evaluation studies were conducted at the USDA-ARS-WRRC quarantine laboratory in Albany, CA and in the field in eastern Turkey.

Insects for release will be processed through the WRRC quarantine laboratory.

Initial releases and monitoring will be made in California by Lincoln Smith in collaboration with California Department of Food and Agriculture scientists.

Table of Contents

1. Introduction:	4
1.1 Nature of the Problem	4
1.2 Proposed Action.....	4
2. Target Weed Information	5
2.1 Taxonomy	5
2.1 Description.....	6
2.3 Distribution of Target Weed.....	6
2.4 Taxonomically Related Plants	8
2.5 Distribution of Taxonomically Related Plants in North America	9
2.6 Life History.....	10
2.7 Impacts	11
2.8 Alternative Management Options	14
3. Biological Control Agent Information.....	18
3.1 Taxonomy	18
3.2 Geographical Range	19
3.3 Known Host Range (Specificity).....	19
3.4 Life History.....	20
3.5 Population of the Agent Studied	20
4. Experimental Methodology and Analysis	20
4.1 Test Plant List	20
4.2 Design of Host Specificity Experiment.....	25
4.3 Positive Controls.....	27
4.4 Reason for Decisions.....	28
4.5 Design of Impact Experiment.....	28
5. Results and Discussion.....	29
5.1 Host specificity	29
5.2 Impact on target plant.....	31
5.3 Summary of Results	32
5.4 Protocol for Releasing the Agent.....	33
5.5 Post-Release Monitoring	34
5.6 Benefit/Risk	34

6. Potential Environmental Impacts.....	35
6.1 Human Impacts	35
6.2 Potential Economic Impacts	35
6.3 Plant Impacts.....	35
6.4 Nonplant Impacts.....	36
6.5 Proposed Methods for Mitigation	37
6.6 Abiotic and Edaphic Effects	37
6.7 Outcome of No Action	37
7. Petitioner’s Conclusion	38
Acknowledgements.....	39
Literature Cited.....	40
Tables	51
Figures.....	63
Appendices	
Appendix 1. Genera and Higher Taxa in the Family Asteraceae occurring in North America.....	87
Appendix 2. Host Plant Test List (all in family Asteraceae).....	99
Appendix 3. Federally listed Threatened and Endangered species in the family Asteraceae that occur in the continental United States	101
Appendix 4. Key to species of <i>Ceratapion</i> associated with <i>Centaurea solstitialis</i> and some allies (Boris Korotyaev unpubl.)	107
Appendix 5. Fecundity, development and behavior of <i>Ceratapion basicorne</i> (Coleoptera: Apionidae), a prospective biological control agent of yellow starthistle (Smith and Drew in press). Environmental Entomology.....	113
Appendix 6. Assessment of risk of attack to safflower by <i>Ceratapion basicorne</i> (Coleoptera: Apionidae), a prospective biological control agent of <i>Centaurea</i> <i>solstitialis</i> (Asteraceae) (Smith <i>et al.</i> 2006). Biological Control.....	131

1. Introduction:

1.1 Nature of the Problem

Yellow starthistle (YST), *Centaurea solstitialis* L., 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 U.S. and Canada (Duncan 2001, Pitcairn *et al.* in press). Infestations have been reported in 23 of the contiguous 48 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.* in press) and threaten states to the east. 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. Its flowers have inch-long spines that deter feeding by grazing animals and lower the utility of recreational lands. Consumption of YST by horses causes a fatal syndrome known as "chewing disease" or nigropallidal encephalomalacia (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. Yellow starthistle originates from the Mediterranean region, where it 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 introduced already, 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 diseases have been described on YST in California (Pitcairn *et al.* 1999a). 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 combined effect of these natural enemies does not appear to have significantly reduced YST over most of its range (Balciunas and Villegas 1999, Pitcairn *et al.* 2002, Smith 2002). 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. Additional agents are needed, especially ones that attack the foliage, stem, and roots of rosettes and young bolting plants (Smith 2004a).

1.2 Proposed Action

We propose to introduce the rosette weevil, *Ceratapion basicorne* (Illiger) (Coleoptera: Apionidae) from Turkey to the western United States for biological control of yellow starthistle. Initial experimental releases will be made at several sites in the western U.S. to determine release protocols and to measure impact on yellow starthistle in different environments. If the weevil becomes well established at experimental release sites and impacts the weed, then we will make it available for widespread release in the western U.S. (following the Code of Best Practices; Balciunas 2000).

2. Target Weed Information

2.1 Taxonomy

Full classification (Bremer 1994, Bremer *et al.* 1999, Brands 2004):

Division: Magnoliophyta
 Class: Magnoliopsida
 Subclass: Asteridae
 Order: Asterales
 Family: Asteraceae
 Subfamily: Cichorioideae
 Tribe: Cardueae
 Subtribe: Centaureinae
 Genus: *Centaurea*
 Subgenus: *Solstitiaria*
 Species: *solstitialis*

Common names: Yellow starthistle, St. Barnaby's thistle

Scientific name: *Centaurea solstitialis* L.

Synonyms: *Calcitrapa solstitialis* (L.) Lam.

Centaurea erythracantha Halacsy

Centaurea sicula L. subsp. *schouwii* (DC.) Nyman

Morphological variation of *Ce. solstitialis* in Eurasia has resulted in the description of six subspecies (Table 1, Wagenitz 1975, Dostál 1976). Fred Hrusa, California State Botanist, examined specimens from several populations in California and found characteristics that suggest the presence of at least three subspecies: *Ce. s.* subsp. *schouwii*, *Ce. s.* subsp. *erythracantha*, and *Ce. s.* subsp. *solstitialis* (in the sense employed in Dostál 1976). These variants occur randomly throughout the state. Our taxonomic treatment of *Ce. solstitialis* for this petition is in a broad sense (*sensu lato*) and includes all the subspecies listed above.

Voucher specimens of California yellow starthistle used in the host specificity tests are deposited at USDA-ARS Foreign Disease-Weed Science Research Unit, Fort Detrick, MD, USDA-ARS Exotic Invasive Weeds Research Unit, Albany, CA, and the herbarium of the California Department of Food and Agriculture (CDA), Sacramento, CA.

The genus *Centaurea* is distributed from Spain across southern Europe to Turkey and Iran and historically has contained 200 to 600 species depending on the taxonomic treatment (Klokov *et al.* 1963, Hellwig 2004). The *Flora Europaea* lists 221 species (Dostál 1976), the *Flora of Turkey* lists 172 species (Wagenitz 1975) and the *Flora URSS* lists 178 species (Klokov *et al.* 1963). The genus is taxonomically difficult, and its phylogenetic relationships are not completely resolved (Garcia-Jacas *et al.* 2001). Several genera have recently been included in the genus (*e.g.*, *Cnicus*) whereas other taxa have been split off as distinct genera (*e.g.* *Plectocephalus*, which includes 2 North American species) (Garcia-Jacas *et al.* 2000, 2001). *Centaurea solstitialis* was placed in the section *Mesocentron* by Wagenitz (1975) and in the subgenus *Solstitiaria* by Dostál (1976).

2.1 Description

Yellow starthistle is a herbaceous winter annual that is usually 0.1-1.0 m (4-40 in.) tall at maturity, depending on growing conditions (Fig. 1; Keil and Turner 1993, Roché and Roché 2000). It has a vigorous tap root that grows 1-3 m (3-9 ft.) deep. Stems are upright, stiff, winged and branched. Leaves are somewhat scabrous or bristly. Leaves of the rosette and lower stem are lobed (5-15 cm long); leaves of the upper stem and flower are long and narrow, with a wing-like appendage running down the stem. Composite flowerheads (capitula) occur singly at the ends of branches and have a long sharp spine (10-25 mm; 3/8-1 in.) extending from each bract giving the flowerhead a star-shaped appearance. Mature flowerheads are 3-17 mm in diameter and consist of many small yellow flowers (10-100) with corollas 13-20 mm long. Each flower is capable of producing one seed, except for the sterile ray flowers on the outer edge of the flowerhead. Yellow starthistle produces both pappus-bearing and non-pappus-bearing seeds. Taxonomic keys to identify the plant are available in Roché and Roché (2000) and Keil and Turner (1993).

2.3 Distribution of Target Weed

Yellow starthistle is native to Eurasia, probably originated 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). Examination of regional floras of Europe and Asia indicates that 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 (Fig. 2; Komarov 1934, Klokov *et al.* 1963, Wagenitz 1975, Dostál 1976). More subspecies of YST (see "Genetic Variability" below) occur in or near Turkey than in any other region, indicating that this is the center of diversity. Yellow starthistle is a frequent casual 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 wet winters and dry summers (Mediterranean climate).

Yellow starthistle has spread to South Africa, Australia, Chile, and the United States (Fig. 3). 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 2002). However, it is most abundant in California, Oregon, Washington and Idaho (Fig. 3; Sheley *et al.* 1999, Duncan 2001). A 1997 survey by the California Department of Food and Agriculture (CDFA) found YST in 42% (n=1935) of California's 4,638 townships (6x6 miles), and the infestations were reported as "high" in 22% (n=1,019) of the townships (Pitcairn *et al.* 1998). In North America, YST 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 spread explosively during the last twenty years (Fig. 4). A 2002 survey in California indicated that the area infested by the weed has increased by 81% since 1985 (Pitcairn *et al.* in press). YST 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.

Genetic Variability

Six subspecies of *Centaurea solstitialis* have been described in Eurasia based on morphological characters (Table 1; Wagenitz 1975, Dostál 1976). The subspecies *Centaurea solstitialis solstitialis* occurs throughout most of the species' range: from Spain to Turkey, Armenia, and Iran (Dostál 1976). The other subspecies have restricted or endemic distributions in Eurasia: *Ce. s. schouwii* occurs only in Sicily and Sardinia; *Ce. s. erythracantha* in eastern Greece; and *Ce. s. adamii* in the central and eastern Mediterranean region. The subspecies *Ce. s. carneola* is endemic to southern Turkey, and *Ce. s. pyracantha* is endemic near Antalya, Turkey (Wagenitz 1975). Thus, more subspecies occur in or near Turkey than in any other region. Fred Hrusa, California State Botanist, examined specimens from several populations in California and found characteristics that suggest the presence of at least three subspecies: *Ce. s. subsp. schouwii*, *Ce. s. subsp. erythracantha*, and *Ce. s. subsp. solstitialis* (in the sense employed in Dostál 1976). These variants occur randomly throughout the state.

Early studies of morphological characteristics of yellow starthistle collected from Oregon, Washington, Idaho and California found significant differences among populations, but the differences were not correlated with location or precipitation zones (Roché 1965, Sheley *et al.* 1983a, b). However, analysis of seed protein banding (isozymes) in 13 different populations of yellow starthistle collected in Washington, Idaho, and California, showed no significant differences (Schumacher *et al.* 1982). Further research with isozymes in the western United States showed that high levels of genetic diversity exist within YST populations, suggesting that multiple introductions may have occurred (Sun 1997). The level of genetic diversity was similarly high in all populations tested, with little interpopulation divergence. Thus, YST in the western U.S. appears to be one highly mixed population of genetically diverse individuals. The genetic variability of this species worldwide is currently under investigation using more advanced molecular genetic tools (D. Luster, USDA-ARS, Ft. Detrick, MD, personal communication). For this petition, we will use the name *Ce. solstitialis* in the broad sense (*sensu lato*), to include all these subspecies and variants.

Habitats Occupied by the Weed

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 m (7,000 ft) (Pitcairn *et al.* 1998, in press). YST 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 mm (10-60 in.). 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. YST 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).

2.4 Taxonomically Related Plants

Yellow starthistle is in the sunflower family (Asteraceae), which is the largest plant family and contains over 23,000 named species (Bremer 1994). Taxonomic relationships within the family are complex and not completely understood (Fig. 5). This proposal will follow the work of Bremer (1994) and Susanna *et al.* (1995) for the taxonomy of the subfamilies, tribes, and subtribes.

Yellow starthistle is in the subfamily Cichorioideae. According to cladistic work by various authors, the tribes of the subfamily Cichorioideae appear to be a paraphyletic grade, with the subfamily Asteroideae being a monophyletic group pulled from among their ranks and raised to subfamily status (Fig. 6). This means that the different tribes of the Cichorioideae may be no more closely related among themselves than they are to the Asteroideae. The apparent relationships among the tribes of Cichorioideae and the Asteroideae vary, depending upon the character set that is used to study the relationships. The tribe Cardueae, however, appears to be a monophyletic group that clearly segregates from the other Cichorioideae tribes and the Asteroideae subfamily (Garcia-Jacas *et al.* 2002). In several classifications, summarized by Bremer (1994) (Fig. 6), the tribe Cardueae segregates out as an isolated basal group with no close relatives, and there is no consistent agreement as to its closest relative.

The exact taxonomic relationships within the tribe Cardueae are not completely understood. Bremer (1994) (Fig. 7, based on morphological data) and Susanna *et al.* (1995) (Fig. 8, based on DNA sequence data) provide two somewhat differing descriptions of the relationships within the tribe. Nevertheless, broad patterns emerge, with the subtribe Centaureinae segregating as a monophyletic group from the rest of the tribe. The two studies did not use exactly the same taxa, which causes some difficulty in making comparisons, but for all the taxa that appear in both trees, the trees agree on their subtribe placement. For example, the genera *Centaurea*, *Carthamus*, *Serratula*, *Volutaria*, and *Stemmacantha* appear in the Centaureinae in both treatments, while *Jurinea*, *Galactites*, *Cynara*, and *Cirsium* are more distantly related and are placed in the subtribe Carduinae. Recent genetic analysis further supports the monophyly of the subtribe Centaureinae, but the subtribe Carduinae is a paraphyletic grade (Garcia-Jacas *et al.* 2002). So, genera in the Carduinae may be no more closely related among themselves than they are to those in the Centaureinae. The two treatments (Bremer 1994 and Susanna *et al.* 1995) disagree on the genetic distance of the different Carduinae taxa from the Centaureinae. The two studies also do not always agree on the relationships within the different subtribes. For example, Bremer shows *Volutaria* more closely related to *Centaurea* than *Carthamus*, whereas Susanna *et al.* suggest the reverse. It is notable that only the clades representing Centaureinae and Cardueae have high bootstrap values, so all other relationships lack sufficient statistical support and it is not surprising that such unsupported hypotheses may disagree. Overall, the differentiation of the Centaureinae seems reasonably clear, while relationships within and between subtribes are more uncertain. The *Arctium-Cousinia-Saussurea-Jurinea* group is now considered to be the closest related group to Centaureinae (Hellwig 2004).

The two subtribes Centaureinae and Carduinae have distinctly different secondary chemical compounds, which are probably important in determining herbivore host plant specificity (Susanna *et al.* 1995). Centaureinae produce acetylene aldehydes, chlorhydrins, and acetates, germacrolide-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, gainolide-type

sesquiterpenoids, monomethoxylated flavonoids, and simple cinnamic acids and their derivatives.

The genus *Centaurea* is very large, with 200-600 species, and its definition and extent is in dispute (Klokov *et al.* 1963, Dostál, J. 1976, Susanna *et al.* 1995, Garcia-Jacas *et al.* 2000, Hellwig 2004). The species within the genus are divided into 24 to 41 sections, depending upon the author. The sections began as different genera in Cassini's original treatment of the group, but later in the 19th century Bentham, and later Hoffman, combined them to form the genus *Centaurea* (Bremer 1994). Several genera such as *Amberboa*, *Mantiscalca*, *Stemmacantha*, and *Volutaria* were removed from *Centaurea*, where they were once sections. However, some groups within the genus *Centaurea* still appear to be phylogenetically more distinct than other well recognized genera (Figs. 9 and 10; Garcia-Jacas *et al.* 2001, J. Gaskin unpubl. data). Two North American species, *Centaurea americana* Nutt. and *Ce. 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. Although many of the phylogenetic relationships among the remaining species within the genus *Centaurea* are not precisely known, some groups have been clearly described. The strongest grouping reflects differences in pollen structure (Wagenitz 1955) and DNA nucleotide base sequences (Susanna *et al.* 1995, J. Gaskin unpubl. data). Yellow starthistle is in the *Jacea* group, which is monophyletic and includes many of the other weedy species adventive to North America (Table 2, Figs. 9 and 10).

2.5 Distribution of Taxonomically Related Plants in North America

Many plants in the family Asteraceae are cosmopolitan, and relatives from the tribe Cardueae are commonly distributed throughout the range of YST. 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 1998). 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 YST.

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 holarctic 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. The USDA Plants Database (USDA-NCRS 2002) lists 101 native species and subspecies of *Cirsium* occurring in the United States. The distribution of each species by geographic region was estimated by breaking the United States into 12 regions. The number of *Cirsium* species occurring in each of these regions is shown in Table 3. 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 (E, T): *Ci. fontinale* ssp. *fontinale* (E), *Ci. fontinale* ssp. *obispoense* (E), *Ci. hydrophilum* var. *hydrophilum* (E), *Ci. loncholepis* (E), *Ci. pitcheri* (T), and *Ci. vinaceum* (T). A total of 20 native species, including *Ci. f.* ssp. *fontinale* and *Ci. f.* 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. Phylogenetic relationships among some of these species is presented in Fig. 11 (Kelch and Baldwin 2003).

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).

2.6 Life History

Yellow starthistle is a winter annual in California, with seeds germinating soon after the onset of winter rains, in October to December (Maddox 1981, Roché and Roché 2000). Although most seeds germinate in the fall, some additional germination occurs during the winter and spring, though these plants are less likely to survive and produce seed (Joley *et al.* 1992). In regions with snow, more germination may occur in the spring, but live rosettes have been found surviving under snow. Rosettes appear to grow slowly during the winter and spring, but they establish a deep root system (at least 3 feet deep; DiTomaso *et al.* 2003a). During the dry summer months, these deep roots allow YST to use soil moisture that is not available to most grasses and other native rangeland vegetation (Enloe *et al.* 2004). This permits YST plants to continue growing and producing seed after most other plants have senesced. YST plants "bolt" in late spring to early summer to form flower stalks. Time of bolting depends on latitude, starting in April in California's San Joaquin and Sacramento Valleys, and in May to June in Idaho, which is further north and higher elevation (Maddox 1981). Bolting plants send up an elongated, highly branched stalk that produces spiny flowerheads at the branch terminals. Flowering begins in May to late June and continues through October, depending on the availability of moisture and absence of hard frosts.

Yellow starthistle reproduces entirely by seed (achenes). Seed production is very high, ranging from 10-100,000 seeds per plant, depending on soil moisture and competition. Estimates of seed production in heavily infested areas range from 20-100 million seeds per acre (Maddox 1981, Callihan *et al.* 1993, DiTomaso *et al.* 1999). Yellow starthistle is an obligate out-croser that produces very little seed unless it is pollinated by insects (Maddox *et al.* 1996). An individual flowerhead is open for 4-5 days, after which the yellow petals fade and seeds develop in 8 or more days (Benefield *et al.* 2001, M. Pitcairn pers. comm.). At maturation, the flowerhead turns brown and dry, the flower petals fall out and seeds disperse onto the soil. Two kinds of seeds are produced: the central seeds, which have pappus hairs, and the outer seeds, which are black and have no pappus. The pappus seeds leave the flowerhead soon after

maturation, usually 2-3 weeks after flowering. The black, non-pappus seeds remain in the flowerhead until late winter when the flowerhead breaks down from the effects of wind and rain.

Non-pappus seed initially tends to be more dormant than pappus seed. The pappus is not very effective for wind dispersal and about 90% of seed falls within 2 ft of the plant (Roché 1965). However, the pappus may help seed stick to animal fur, and pappus seeds are easily ejected from the dry flowerhead when the plant is bumped. Human activities, including seed contaminated mud on vehicles, contaminated crop seed or hay, contaminated soil used for construction, and transporting livestock from infested fields, can also contribute to rapid and long distance spread of the seeds.

Although almost all YST seeds are capable of germinating soon after maturation (Joley *et al.* 1997, 2003, Roché *et al.* 1997, Benefield *et al.* 2001), they usually do not because of the absence of summer rain and because germination is quickly inhibited by typical warm summer temperatures (above 30°C). Natural exposure to heat lowers the germination threshold temperature, which then gradually increases with age. Thus, seeds are inhibited from germinating in the summer but become more responsive as fall progresses and temperature drops. In California, most seeds near the soil surface germinate soon after the onset of fall rains in October or November. Seeds that have been buried too deeply remain dormant until the soil is disturbed, exposing the seeds to light, which helps break dormancy. Although buried seeds can persist for up to ten years under some field conditions (Callihan *et al.* 1989, 1993), the number of viable seed near the soil surface can decrease rapidly with time because of germination and destruction by arthropods and microbes. After four years in California, less than 1% of the original seeds remained ungerminated and still viable (Joley *et al.* 2003).

In suitable habitats in the western U.S., yellow starthistle can produce much more seed than is necessary to maintain the population the following year. In many areas, dense populations of seedlings occur, and often only a small proportion (20%) survive to reach reproductive maturity (Sheley and Larson 1994, Pitcairn *et al.* 1995, 2000). However, this mortality reduces intraspecific competition, allowing yellow starthistle plants to grow larger and produce as much seed per square meter as those in crowded populations (Pitcairn *et al.* 2000).

2.7 Impacts

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, YST displaces desirable forage species and interferes with livestock grazing once the spines develop.

Consumption of YST is toxic to horses (Kingsbury 1964). Continued feeding causes mycosal 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, Akba *et al.* 1995). Yellow starthistle poisoning is generally most dangerous when it 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 YST'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 YST 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 YST uses deep soil moisture reserves earlier than these species, dense YST infestations cause these plants to experience drought conditions even in years with normal rainfall (Gerlach *et al.* 1998). YST water consumption also impacts human economic interests (Dudley 2000). The California State Water Resources Control Board recently 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. Conventional control methods are generally expensive and may pose some risk to wildlife, homeowners, and land managers. For example, prescribed burns can pollute the air, compromise the health of wildlife, and can develop into catastrophic wildfires if they escape. 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, the Defense Department, U.S. 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 was recently estimated by the Agricultural Issues Center, University of California, Davis (Jetter *et al.*, 2003) and is summarized here. 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% 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%, 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 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. Because there are large variations in land quality and the measurement of the economic values of controlling yellow starthistle is difficult, these two benefit levels provide a reasonable range in which to assess the benefits of the biological control program.

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 YST 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 a few purported 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 honeybees 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, YST may actually encourage displacement of the native bees by honeybees. 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 honeybees (Barthell *et al.* 2000). Yellow starthistle also displaces native flowers to which native pollinators are adapted.

Young YST plants have some forage value to cattle before they bolt and develop spines (Callihan *et al.* 1995). Young plants contain between 8 to 14% 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 YST-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 pheasant, mourning dove, California quail, gold finches and house finches) and rodents (Martin *et al.* 1951, Roché 1965). However, seed produced by YST is at the expense of that produced by native forbs and grasses which are displaced by the weed. In general, native wildlife is expected to thrive better with native plants than with introduced plants, and we know of no evidence that shows that YST is more beneficial to native wildlife than native plants. If other wildlife such as deer were found to graze on YST 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 YST

seed, some viable seed can pass through their guts, which increases the risk of dispersing the weed to new locations (Roché 1965).

Successful biological control is expected to reduce the population of YST gradually, which will allow other species of plants and animals time to respond to the change. Reducing YST populations will provide a much greater benefit to ranchers, managers of rights-of-way, recreational use of land, and native plants and wildlife than the possible loss of revenue to those beekeepers who use YST as a nectar source.

2.8 Alternative Management Options

The most successful management strategy is to integrate several control tools and combine them with monitoring to eliminate new infestations before they grow and spread (Enloe *et al.* 1999, DiTomaso *et al.* 2000). Although several methods can effectively kill plants, the residual soil seed bank makes it important to follow-up management for at least 3 years. The major limitations for most of these methods are the economic cost of applying them to large areas of rangeland and environmental restrictions. Combining weed management with general vegetation management will go beyond controlling a specific weed and help result in a desirable plant community and prevent future invasion by other weeds.

Chemical control

Yellow starthistle can be controlled by a number of nonselective pre-emergence herbicides, including simazine, diuron, atrazine, imazapyr, imazapic, metsulfuron, sulfometuron, chlorsulfuron, bromacil, tebuthiuron, oxyfluorfen, and prometone (DiTomaso 2005). All these compounds are registered for use on rights-of-way or industrial sites (although not all in California), but few can be used in rangeland, pastures, or wildlands. In rangeland, only metsulfuron (which is not registered in California), and to some degree chlorsulfuron (not registered for pastures or rangeland in any state), provide selective control of yellow starthistle without injuring most desirable grasses.

Broadleaf-selective post-emergence herbicides are most effective on seedlings, but those registered for use in California, such as 2,4-D, dicamba, triclopyr or glyphosate, do not have residual soil activity. Because yellow starthistle seeds germinate continuously throughout the winter and spring, herbicide must be applied repeatedly. Making only one application at the end of the rainy season is not effective because most plants are too large to be controlled.

The preferred herbicide for yellow starthistle in California is clopyralid, a growth-regulator herbicide. It is registered for use on non-crop areas of California, including pastures, rangeland, and wildlands. It is safe on grasses but will significantly damage composites (Asteraceae), legumes (Fabaceae), nightshades (Solanaceae), and some species in the knotweed (Polygonaceae), carrot (Apiaceae) and violet (Violaceae) families. Control costs are about \$25 per acre. Intense use of clopyralid against yellow starthistle has created concern about possible development of herbicide resistance.

Picloram is the most widely used herbicide to control yellow starthistle in other western states. It has a long residual soil activity, which increases its effectiveness but also the risk of environmental contamination, which is why it is not registered for use in California. Intensive use of picloram in the 1980s led to development of resistance by at least one population of yellow starthistle in Washington state (Callihan *et al.* 1990). This population was also cross-resistant to other auxin-type herbicides, including clopyralid, dicamba and fluroxypyr (Fuerst *et al.* 1994, Valenzuela-Valenzuela *et al.* 1997).

Mechanical control

Mowing can be effective when timed appropriately and repeated several times during a season (Thomsen *et al.* 1994, 1997, Benefield *et al.* 1999). To be effective, mowing must cut the plants after they have bolted, which minimizes resprouting, but before they set seed. It is also most effective if there is competing vegetation that causes YST to be more erect and high-branching. The method is limited to flat, relatively smooth ground, particularly along roadways and in recreational areas. Mowing is not always successful, and it can injure late growing native forb species, and reduce fall and winter forage for wildlife and livestock (DiTomaso *et al.* 2000). Mowing too early results in prostrate plants that still flower and produce seed.

Hand pulling individual plants before they flower is effective in small populations, but plants will regrow if the stem is not completely removed (DiTomaso 1997). Hand pulling can disturb the soil, which creates ideal establishment sites for seedlings of YST or other weeds. Frequent scouting-and-removal campaigns are necessary throughout the growing season (about every 2-4 weeks). Without volunteer crews, this is very expensive and too impractical to implement over large areas.

Tillage, using plows or discs, in the late spring or early summer before the plant flowers, will control yellow starthistle, provided that the roots are detached from the shoots, and the surface soil is dry enough to prevent regrowth from fragments (DiTomaso 1997). This method is practical primarily only in cultivated fields and on some roadsides.

Cultural control

Cultural practices can be effective for yellow starthistle control, but they have severe limitations. Often, the timing or intensity of the control effort is crucial, and can mean the difference between decreasing or increasing the YST population.

Prescribed burning has produced mixed results. While burning has resulted in some successful control efforts (Hastings and DiTomaso 1996, DiTomaso *et al.* 1999a), Sheley *et al.* (1999) reported that burning was ineffective. The best time for burning is usually in early to mid-summer (June to early July), after YST 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 YST is still green at this season, it is killed by scorching rather than burning, and it does not fuel the fire. Fire has little if any impact on seeds in the soil. Burning can increase plant size and seed production of YST 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 (*i.e.*, high labor costs) to manage the fire and keep it contained. 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.* 2003b).

Livestock grazing can effectively reduce YST 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 YST (Thomsen *et al.* 1996). Intensive grazing in May and June, when YST bolts but before it is spiny, reduced growth, canopy cover, survivability, and reproductive capacity of yellow starthistle (Thomsen *et al.* 1989, 1990, 1993). On the other hand, grazing cattle in late winter or early spring can harm desirable grasses and increase YST infestations. Trampling vegetation also tends to favor YST (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 YST 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.

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). 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

Several species of endemic natural enemies attack yellow starthistle in North America, including fungal pathogens that kill seedlings (*Ascophyta* n. sp., *Sclerotinia minor*, and *Colletotrichum gloeosporioides*; Pitcairn *et al.* 1999a, Woods *et al.* 2000) sometimes causing up to 80% mortality. However, the presence of any single disease was sporadic or somewhat localized. The European grey garden slug, *Deroceras reticulatum* causes similarly sporadically high mortality to seedlings (Pitcairn *et al.* 2000). Arthropod herbivores occasionally found on YST include some typical generalists: aphids, cicadellids, fulgorids, mirids, thrips and spider mites. Larvae of the native painted lady butterfly (*Vanessa cardui*) feed on leaves, and the exotic beetle, *Lasioderma haemorrhoidale*, is relatively common in mature flowerheads. A stem-mining mordellid beetle occurred at 4 of 5 sites. However, no root feeders and no plant diseases evident in mature plants at end of season were found. High densities of YST persist despite the presence of these generalist natural enemies.

A classical biological control program for YST began in the 1960s (Turner *et al.* 1995, Piper 2001, Pitcairn *et al.* 2004). A total of 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 (Table 4). The fly *Urophora jaculata* was approved for release but failed to establish because it was not adapted to the North American population of YST (Clement 1994). Another fly *Chaetorellia succinea* was accidentally introduced in 1991 and has become well established (Balciunas and Villegas 1999). The host specificity of this fly was subsequently evaluated, and it was found to pose a small risk to safflower, no risk to native North American *Cirsium* species, and possible risk to two native North American *Centaurea*

species (Balciunas and Villegas 2001, Balciunas pers. com.). However, the latter two plants occur outside the current geographic distribution of YST, which lowers their chance of being attacked.

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; E. Coombs, G. Piper and T. Prather pers. com.). *Bangasternus orientalis* and *U. sirunaseva* are widespread in California, but not very abundant (< 25% of flowerheads infested). *Larinus curtus* and *Ch. australis* are neither widely established and nor abundant in California, although they are doing better 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 (Pitcairn pers. com.). The peacock fly, *Ch. australis*, emerges early in the spring, long before yellow starthistle flowerheads are available for oviposition. 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 YST 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 YST are better synchronized.

Surveys conducted in California show that each of the flowerhead insects are most abundant in different environmental regions (Pitcairn *et al.* 2003, unpubl. data). 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. As data become available from other states, we will have a clearer understanding of the environmental constraints for each species.

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% of flowerheads, depending on year and site. At some sites YST 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 YST density (E. Coombs pers. com.).

The exotic rust pathogen, *Puccinia jaceae* var. *solstitialis* was approved for release in California in 2003 and was released in 20 counties in 2004 (Woods and Villegas 2004, Woods *et al.* 2004a). Although infection occurred at all sites, the rust has not yet shown signs of spreading very far. Experimental releases are being continued in California, as are experiments to measure impact and dispersal. The rust is expected to reduce the size and seed production of YST but not to cause much plant mortality (Shishkoff and Bruckart 1996).

It appears that additional biological control agents are needed to control the weed over most of its geographic range (Balciunas 1998, Smith *et al.* 2001, Uygur *et al.* 2004). There is especially a need for agents attacking the root, stem, and leaves of rosettes before they bolt and flower. It is expected that natural enemies stressing the immature plants may increase mortality and/or substantially reduce the number of maturing flowerheads available for attack by the established flowerhead insects. The combination of the attack during the growing and flowering phases should increase the level of control of this serious weed.

3. Biological Control Agent Information

3.1 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. We propose using the common name, "yellow starthistle rosette weevil". 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:	Ceratapiini
Genus:	<i>Ceratapion</i>
Subgenus:	<i>Echinostroma</i>
Species:	<i>basicorne</i>

A detailed modern description of the species and taxonomic keys for genera in the Tribe Ceratapiini and species in the Genus *Ceratapion* are available (Alonso-Zarazaga 1990a, Wanat 1994). Boris Korotyaev (unpubl. data, Appendix 4) made detailed illustrations and a key to species of *Ceratapion* that have been found on yellow starthistle.

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. The center of origin of the Ceratapini is probably in western and central Asia, coinciding with that of their Asteraceae host plants (*Artemisia*, *Anthemis*, *Echinops*, tribe Cardueae, etc.). Most speciation within the genus occurred in the Pliocene-Miocene, and sibling species appeared in the Pleistocene Glacial Period. This coincides with the time when the major modern Cardueae clades (*Centaurea*, *Colymbada* group, *Cyanus* and *Carthamus/Carduncellus* group) are also thought to have originated (Hellwig 2004). *Ceratapion basicorne* is in the subgenus *Echinostroma*, and is probably closest related to *C. curtii* and *C. penetrans* followed by other members of the subgenus, including *C. scalptum* (Fig. 12; Alonso-Zarazaga 1990a, Wanat 1994). The nearest subgenera to *Echinostroma* are *Acanephodus* and *Clementiellus*, which contain *C. onopordi* and *C. orientale*.

Specimens of *Ceratapion basicorne* from our colony in quarantine were identified by B.A. Korotyaev (Russian Acad. Sci., St. Petersburg), who is a taxonomic specialist of Palaearctic Curculionidae. Methods of identification using DNA analysis are currently being developed by M. Cristofaro and G. Antonini (ENEA C.R. Casaccia, Rome, Italy). Voucher specimens have been deposited at the California Department of Food and Agriculture in Sacramento, CA, and at the USDA Systematic Entomology Laboratory (SEL) in Beltsville, MD.

3.2 Geographical Range

Ceratapion basicorne is distributed throughout Europe and southwestern Asia, from Spain to Azerbaijan, between 37° and 55° N latitude (Fig. 13, 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 (Figs. 2 and 3). 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, J. Balciunas unpubl. data). If *C. basicorne* is released in North America, it is expected to become established throughout the range of yellow starthistle.

3.3 Known Host Range (Specificity)

Although *Ceratapion basicorne* has been collected over a wide region, it has been found on very few host plants (Table 5). It has primarily been reared from YST, but there are also reports of rearing it from *Centaurea cyanus* (bachelor's button, cornflower), *Centaurea depressa* (which is very similar to *Ce. cyanus*), and in one case, *Cnicus benedictus*. *Cn. benedictus* has recently been placed in the Jacea group of *Centaurea* (which includes YST), based on phylogenetic analysis of DNA (Garcia-Jacas *et al.*, 2000). Thus, the insect has only been reared from a few species of plants in the Jacea and Cyanus groups, within the genus *Centaurea*. Adults have been found resting on plants only in the Cardueae tribe. There is an anomalous report of *C. basicorne* developing in flowerheads of *Ce. cyanus* (Dieckmann 1977), but based on knowledge of the insect's life history (see below) this appears to be mistaken (Wanat 1994). Perhaps adults hiding in dry flowerheads could account for this observation.

Ceratapion basicorne has been successfully reared from other host plants that were artificially infested by transferring young larvae: *Carthamus tinctorius* (safflower), *Galactites tomentosa*, and *Carduus pycnocephalus* (Clement *et al.* 1989). In the same study, larvae failed to develop on *Centaurea calcitrapa*, *Cnicus benedictus*, *Carthamus dentatus*, *Cynara scolymus*, *Cirsium douglasii*, *Cirsium campylon*, *Zinnia elegans*, and *Lactuca sativa*.

Related taxa

Species in the subgenus *Echinostroma*, which includes *C. basicorne*, feed on plants in the genera *Arctium*, *Carlina*, *Carthamus*, *Centaurea* and *Silybum*, which are all in the tribe Cardueae in the subfamily Cichorioideae (Alonso-Zarazaga 1990a, Wanat 1994). Larval host plants of the closest related species, *Ceratapion curtii* (Wagner), are unknown. Hosts of the next closest related species, *Ceratapion penetrans* (Germar) are: *Centaurea rhenana* [= *stoebe*, = *maculosa*, = *paniculata*], *Ce. jacea*, *Ce. cyanus*, *Ce. diffusa*, *Ce. nigra*, *Ce. scabiosa*, *Ce. solstitialis*, *Arctium lappa*, and *Carlina vulgaris*. *Ceratapion scalptum* (Mulsant & Rey), which is also in the same subgenus, attacks *Carthamus* and *Silybum* species. *Ceratapion orientale* (Gerstaecker) (subgenus *Clementiellus*) has been reared from only *Centaurea rhenana*, whereas *C. onopordi* (Kirby) (subgenus *Acanephodus*) attacks many species of *Arctium*, *Carduus*, *Cirsium*, *Centaurea*, *Onopordum*, and *Cnicus*. *Ceratapion damryi* (Desbrochers) (subgenus *Ceratapion*), is a well known pest of artichoke (*Cynara scolymus*), and its larval hosts are only in the genus

Cynara. 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: *Xeranthemum*, *Echinops*, *Carduus*, *Cirsium*, *Galactites*, *Onopordum*, *Cynara*, *Silybum*, *Centaurea* and *Arctium*.

Several species in the family Apionidae have previously been used for biological control of weeds (Julien and Griffiths 1998). *Exapion ulicis* (Forster) was introduced to western U.S. in 1953 to control gorse (*Ulex europaeus*; Fabaceae) (Coombs *et al.* 2004). The weevil became well-established in California, Oregon and Washington and attacks 30 to 90% of seed pods. *Apion fuscirostre* F. was introduced to control Scotch broom (*Cytisus scoparius*; Fabaceae) in the U.S. in 1964 (Andres and Coombs 1995). It became established in California, Oregon and Washington and destroys about 60-85% of seed. *Perapion* (= *Apion*) *antiquum* (Gyllenhal) was introduced to Hawaii to control *Emex spinosa* (L.) Campd. (devil's thorn, lesser Jack; Polygonaceae) in 1957 and provides complete control at 600 to 1200 m elevation (Krauss 1963).

3.4 Life History

Ceratapion basicorne adults emerge from hibernation in the early spring and feed on yellow starthistle leaves (Fig. 14; Clement *et al.* 1989, Smith and Drew in press, Appendix 5). 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 YST leaves for a few days then disappear. They are thought to aestivate and hibernate in secluded places, and adults have been found under tree bark in July (Hayat *et al.* 2002). Newly emerged females are in reproductive diapause, 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 (Smith and Drew in press).

No pathogens of *C. basicorne* are known, but some chalcidoid parasitoids have been found in roots of YST apparently infested by larvae. *Trichomalus* sp. aff. *gynetelus* Wek. (Hymenoptera: Pteromalidae) was collected near Bingol, Turkey on 3 June, 2001 by L. Gultekin. However, parasitism rates in Turkey appear to be very low (less than 5%).

3.5 Population of the Agent Studied

One mixed population of the weevil was studied in the USDA-ARS quarantine laboratory in Albany, CA. Yellow starthistle plants infested with apionid larvae and pupae were collected at sites near Kayseri, Sivas, Erzincan, Erzurum, and Malatya, Turkey between June 4 and 8, 2001 (Smith & Drew in press, Appendix 5). Adults were reared from these plants inside quarantine and were individually identified by L. Smith before establishing a colony. Identification of representative specimens was confirmed by B.A. Korotyaev. The colony has been maintained in quarantine on YST plants for over 4 years.

4. Experimental Methodology and Analysis

4.1 Test Plant List

The approach used to select the test plants employs several factors. It is based upon the accepted phylogenetic approach outlined by Wapshere (1974), where more species are tested in taxonomic

ranks closely related to the target species, and the number of test species decreases as relatedness to the target decreases. This approach has a well-supported safety record (Pemberton 2000, Sheppard *et al.* 2005). Other factors that contribute to the choice of test species include: nativity in North America; ornamental or other economic value; whether the species is sympatric with the target's present or potential range in the U.S.; 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. We have reviewed the host test lists for *Puccinia jacea* var. *solstitialis* (YST rust; Petition No. 00-07) and for agents of *Acroptilon repens* (Russian knapweed; Petition No. 97-03 as amended and Petition No. 03-02), which is closely related to YST, to assist us in selecting test species. Species names and number of taxa in North America are based on the PLANTS Database (USDA-NRCS 2002) with the support of other regional flora, primarily Barkley (1986), Keil (1993) and Hitchcock and Cronquist (1998).

Category 1: Genetic types of the target

Although there is substantial genetic variation among yellow starthistle plants, there are no distinctly separate populations occurring in the United States (see "Taxonomy" section). Therefore, we used seed collected primarily from one location, in Alameda county, California.

Category 2: North American species in the same genus

General considerations:

All but two of the North American species in the genus *Centaurea* are non-native, and many of them are noxious weeds (see "Taxonomy" section). A biological control agent that damages such species is more a benefit than a cause for concern. Because *Centaurea* is a large and diverse genus, we tested species in several different taxonomic "Sections" defined primarily by the different pollen types (Table 2), in order to determine intrageneric specificity of the candidate agent. The two native species, *Ce. americana* and *Ce. rothrockii* (Sect. *Plectocephalus*), which are not considered rare, were tested to determine if they are at risk. These two natives were recently assigned to the genus *Plectocephalus*, and are distantly related to yellow starthistle (Wagenitz 1955, Hellwig 2004). We also tested three species considered to be ornamentals: *Ce. cyanus* (bachelor's button, cornflower) and *Ce. montana* L. (Sect. *Cyanus*) and *Ce. cineraria* Jacq. ex Nym. (Sect. *Pannophyllum*). However, *Ce. cyanus* is also an invasive weed in some regions of North America and is a common weed in wheat in Eurasia. *Cyanus* has recently been recently segregated as a separate genus (Greuter 2003).

Category 3a: Other North American species in the same subtribe (Centaureinae)

Native species: There are no other native species of *Centaureinae* in North America.

Non-natives of economic value: We tested ten varieties of safflower (*Carthamus tinctorius*), including both oleic and linoleic varieties: C44 (Cargill); CW88-OL (CalWest Seed Co., oleic, popular in California), CW1221 and CW4440 (linoleic, popular in California); S317, S345, and S518 (SeedTec Seed Co., oleic, popular in California); S555 and S730 (SeedTec Seed Co., linoleic, popular in California); and Gila (adapted to Arizona).

Other non-natives: We tested *Acroptilon repens* (Russian knapweed), *Cnicus benedictus* (blessed thistle) and *Crupina vulgaris* (common crupina), which represent other groups of

Centaureinae in the North America. *Cnicus* has recently been merged into *Centaurea* (Greuter 2003).

Category 3b: Other North American species in other subtribes of the tribe Cardueae (= subtribes Carduinae, Carlininae, and Echinopsidinae)

Native species: The genus *Cirsium* (Carduinae) contains many native species in North America, particularly west of the Rocky Mountains (Table 3). Recent genetic studies moderately support the theory that the North American species form a monophyletic group (quartet puzzling value = 48), suggesting that they all evolved from a common ancestor that originated in Eurasia (Kelch and Baldwin 2003; Fig. 11). There is strong evidence for a well-defined clade endemic to California (quartet puzzling value = 78), but not enough evidence to clearly define any other groups. Two of our test species represent the endemic California group: *Ci. fontinale* and *Ci. hydrophilum*, and the others represent other more weakly defined North American groups: *Ci. brevistylum*, *Ci. ciliolatum*, *Ci. cymosum*, *Ci. loncholepis*, *Ci. occidentale*, and *Ci. vinaceum*. *Ci. vulgare* was included as a Eurasian representative. This list includes several rare or protected species: *Ci. ciliolatum*, *Ci. fontinale* var. *fontinale*, *Ci. hydrophilum* var. *vaseyi*, *Ci. loncholepis*, and *Ci. vinaceum* (Appendix 2).

In the rest of the tribe Cardueae, the only other natives are in the genus *Saussurea* (Carduinae). There are three species in the contiguous U.S. (five others occur in Canada and Alaska, but are beyond the range of YST) (Moore and Frankton 1973). We tested *Saussurea americana*, which is a rare plant occurring in northern California at high elevation (1700 m), but is relatively widespread in the Pacific Northwest.

Non-natives of economic value: The Carduinae include the commercial crop globe artichoke (*Cynara scolymus*), which we tested. Ornaments in the group include *Xeranthemum* (Carlininae) and *Echinops* (Echinopsidinae), so we tested *Echinops exaltatus* and *Xeranthemum cylindraceum*.

Other non-natives in the U.S.: We tested *Carduus pycnocephalus* (Italian thistle, which is more similar to YST in size and geographic distribution than musk thistle, *Ca. nutans*), *Silybum marianum* (milk thistle) and *Onopordum acanthium* (Scotch thistle) in the Carduinae. The only member of the Carlininae in N. America is *Carlina vulgaris*, but it only occurs in the northeastern U.S., beyond the distribution of YST, so we did not test it.

Category 3c: Other North American species in other tribes of the Asteraceae

SUBFAMILY CICHORIOIDEAE

The subfamily Cichorioideae, in which the target weed occurs, is paraphyletic (Fig. 5), so some of the tribes within the Cichorioideae may be no more closely related among themselves than they are to the subfamily Asteroideae. It is uncertain which tribes are most closely related to the tribe Cardueae. We therefore tested at least one species from each tribe (including those within Asteroideae) that has North American species that are either native or that have economic value, and tested more species from tribes that are especially well represented by North American natives or by economic species (Appendix 1).

The tribes of the Cichorioideae that have North American natives are Mutisieae, Lactuceae, and Vernonieae (Bremer 1994). The tribe Mutisieae has about 970 species, but only six of its 76 genera have representatives in North America. The Lactuceae have about 1550 species in 98 genera and are well represented in North America. The Vernonieae have about 1300 species in 98 genera, but they are largely tropical, and only the genera *Stokesia* and *Vernonia* have species

in North America. Another tribe with some ornamental value is Arctoteae, but it has no North American native species.

We tested the following Cichorioideae species:

Arctoteae: The tribe is native to South Africa but contains some introduced genera of ornamental value, such as *Arctotis* and *Gazania*. We tested *Gazania rigens* (treasure-flower) a cultivated perennial.

Mutisieae native: We tested *Trixis californica* (AZ, CA, NM, TX). Representatives of other genera were difficult to obtain (e.g., *Leibnitzia lyrata* (NM, AZ) and *Adenocaulon bicolor* (CA, ID, MI, MN, MT, ND, OR, SD, WA, WY). The other Mutisieae genera have southeastern distributions and are not likely to be sympatric with YST.

Lactuceae natives: Native genera, scattered among different subtribes, include *Crepis* (=Lagoseris), *Taraxacum*, *Lactuca*, *Sonchus*, *Agoseris*, *Microseris*, *Stephanomeria*, *Atrichoseris*, and *Malacothrix*. We tested *Agoseris grandiflora* (Microseridinae) and *Stephanomeria cichoriacea* (Stephanomeriinae). *Agoseris grandiflora* (bigflower agoseris) is a widespread western species (CA, ID, MT, NV, OR, UT, WA) that occurs in the same habitats as YST and has flowers held on stems above a basal rosette of foliage. *Stephanomeria cichoriacea* is a perennial with a large root crown that forms rosettes, and it occurs on dry rocky slopes in the coastal region of the southern half of California. We also tested a commercial "salad bowl" variety of garden lettuce (*Lactuca sativa*, Lactucinae).

Vernonieae natives: *Vernonia* has about 30 distinct species in the U.S. Although they are all restricted to east of the Rocky Mountains, a few species range into Texas, Colorado, and Montana. *Stokesia* has only one species, *S. laevis* (Stokes' aster), which occurs in the southeast. Both genera have some use as ornamentals and potentially also for their oils. We tested *S. laevis* because of availability.

SUBFAMILY ASTEROIDEAE

The subfamily Asteroideae is very large (just the Astereae and Senecioneae have over 6000 species) and well represented in North America. Tribes of Asteroideae with major genera that have species native to the U.S. include: **Anthemideae:** *Artemisia*; **Astereae:** *Aster*, *Baccharis*, *Chrysothamnus*, *Ericameria* (=Happlopappus), *Erigeron*, *Grindelia*, *Heterotheca*, *Machaeranthera*, *Solidago*, *Townsendia*; **Eupatorieae:** *Brickellia*, *Liatris*, *Ageratina*, *Eupatorium*; **Gnaphalieae:** *Anaphalis*, *Antennaria*, *Filago*, *Gnaphalium*, *Psilocarphus*, *Stylocline*; **Helenieae:** *Adenophyllum*, *Arnica*, *Calycadenia*, *Chaenactis*, *Eriophyllum*, *Gaillardia*, *Helenium*, *Hemizonia*, *Hulsea*, *Hymenoxys*, *Lasthenia*, *Layia*, *Madia*, *Perityle*, *Tagetes*; **Heliantheae:** *Ambrosia*, *Balsamorhiza*, *Bidens*, *Coreopsis*, *Cosmos*, *Echinacea*, *Encelia*, *Helianthus*, *Heliopsis*, *Ratibida*, *Rudbeckia*, *Tithonia*, *Verbesina*, *Viguiera*, *Wyethia*, *Zinnia*; **Plucheeae:** *Pluchea*; **Senecioneae:** *Blennosperma*, *Petasites*, *Psacalium*, *Senecio*, *Tetradymia*.

We tested the following native species:

Anthemideae: *Artemisia californica* (California sagebrush), an important component of the coastal sage scrub community, which is a major plant community especially in Southern California.

Astereae: We tested *Symphyotrichum* (=Aster) *chilense* (Pacific aster), which is a perennial up to 1 m tall, with mostly basal, more or less hairy leaves. It is found along the California coast into British Columbia, in grasslands and marshes below 500 m.

Eupatorieae: *Liatrix punctata* (dotted blazing star) is native in the central section of the U.S., from New Mexico to Montana and Louisiana to Ohio. *Brickellia californica* (California brickellbush) is a perennial subshrub that occurs from California to Oregon, east to Texas.

Gnaphalieae: *Pseudognaphalium* (= *Gnaphalium*) *californicum* (ladies' tobacco) is widely distributed throughout California west of the Sierra crest, extending into Washington and Baja California. It is an annual or biennial up to 85 cm tall, with glandular green leaves along branched stems that bear white to pink, rounded flowers.

Helenieae: Since the Helenieae are very well represented in the West (about 53 genera in California), we tested two species in this group: *Hemizonia minthornii* (Santa Susana tarweed, subtribe Madiinae) is a rare perennial in California with glandular leaves and *Eriophyllum staechadifolium* (seaside woolly sunflower; subtribe Baeriinae) a perennial subshrub, as representatives that were obtainable. *Hemizonia* spp. are annuals or perennials that occur from Arizona through California to Washington and east to Nevada and Idaho. *Eriophyllum* spp. are annuals or perennials that occur from California to Washington and east to Arizona and Montana.

Heliantheae: The Heliantheae are moderately well represented in the West (about 25 genera in California), and we tested two species in this group. *Helianthus annuus* (sunflower, subtribe Helianthinae) is an economically important crop and native plant that occurs throughout the U.S. *Echinacea purpurea* (eastern purple coneflower, subtribe Rudbeckiinae) is a perennial up to 1 m tall with mostly basal, dark green lanceolate leaves and pink-purple flowers in small clusters held above the base on long stems. It is widely found in the Plains states and is commonly used as an ornamental.

Plucheeae: This is a relatively small tribe, primarily distributed in the tropics, that has 12 species in N. America, 10 of which are in the genus *Pluchea*. Only two species occur west of the Rocy Mountains: *Pluchea odorata* (salt marsh fleabane) and *P. sericea* (arrow weed). We are currently testing *P. odorata*.

Senecioneae: Although Senecioneae is a large tribe, it is not very well represented in the western U.S. (9 genera in California). Only *Senecio* and *Tetradymia* have more than two or three species, and only *Senecio* could be considered a large genus, with about 52 species represented. We tested *Senecio cineraria* (silver ragwort, dusty miller) an introduced annual ornamental and *S. vulgaris* (common groundsel) a common annual/biennial weed throughout the U.S.

Category 4: Threatened and endangered species in the same family as the target

There are 63 species and subspecies in the family Asteraceae that are federally listed in the continental U.S. (Appendix 3). However, only 11 occur in the subfamily Cichorioideae (in the genera: *Cirsium*, *Malacothrix*, *Stephanomeria*, *Taraxacum*, and *Vernonia*). We tested the T&E species: *Cirsium fontinale* var. *fontinale*, *Ci. loncholepis*, and *Ci. vinaceum*, and substituted *Cirsium fontinale* var. *fontinale* for *Ci. fontinale* var. *obispoense*, *Cirsium hydrophilum* var. *vaseyi* for *Ci. hydrophilum* var. *hydrophilum*, and *Cirsium brevistylum* for *Cirsium pitcheri*. Because *Cirsium* is in the same subtribe (Centaureinae) as the target weed, we also tested *Ci. ciliolatum*, *Ci. cymosum*, and *Ci. occidentale* var. *venustum*. We substituted *Agoseris grandiflora* for *Malacothrix indecora* and *M. squalida*, *Stephanomeria cichoriacea* for *Stephanomeria malheurensis*, *Lactuca sativa* for *Taraxacum californicum*, and *Stokesia laevis* for *Vernonia proctorii*.

For threatened and endangered species in the subfamily Asteroideae, we tested substitute species in the genera: *Artemisia*, *Brickellia*, *Echinacea*, *Eriophyllum*, *Helianthus*, *Hemizonia*,

Liatris, *Senecio* and *Symphyotrichum* (=Aster); with at least one representative for each tribe that has T&E species.

Category 5: Species in other families in the same order as the target

No other species have been identified that have similar morphological or chemical characteristics to those of yellow starthistle.

Category 6: Species in other orders

No other species have been identified that have similar morphological or chemical characteristics to those of yellow starthistle.

Category 7: Plants on which the agent or its close relatives have been reported

Host plants from which *C. basicorne* has been reared include *Centaurea solstitialis* (yellow starthistle), *Ce. cyanus* (bachelor's button), *Ce. depressa*, and *Cnicus benedictus* (blessed thistle) (Table 5). *Centaurea cyanus* and *Cn. benedictus* are alien species that occur in N. America, so we tested them. Host plants of other species in the genus *Ceratapion* (subgenus *Echinostroma*) (see section "Related taxa", above) include plants in the genera *Arctium*, *Carlina*, *Carthamus*, *Centaurea* and *Silybum*, which are all in the tribes Cardueae and Carlineae of the subfamily Cichorioideae. *Ceratapion damryi*, which is in a different subgenus from *C. basicorne*, is a well known pest of artichoke (*Cynara scolymus*). We tested the commercial plants artichoke (*Cy. scolymus*) and safflower (*Carthamus tinctorius*), and the alien weed *Silybum marianum* (milk thistle), which is a common within YST's North American range.

4.2 Design of Host Specificity Experiment

Host specificity of the weevil was determined by use of a series of tests: 1) laboratory no-choice oviposition and larval development, 2) laboratory choice oviposition, 3) field attack rate. The first two experiments were conducted in the USDA-ARS quarantine laboratory in Albany, CA. The field experiments were conducted in eastern Turkey by cooperators (led by M. Cristofaro, ENEA C.R. Casaccia, Rome, Italy and R. Hayat, Ataturk University, Erzurum, Turkey).

1) laboratory no-choice oviposition and larval development.

Individual mated females that had completed reproductive diapause were held in a sealed plastic tube with a cut leaf of yellow starthistle until they oviposited. Each female was then placed in a plastic tube (3.5 cm diam. x 11 cm tall) mounted on rosette leaf of a nontarget plant species for 5 days (Fig. 15). Afterwards, we put each female back on YST for 2-3 days to feed and demonstrate that they can still oviposit. If the female failed to oviposit on YST or died during the experiment, then the preceding replicate was considered invalid, and the plant was retested using another insect. A different female was used for each replicate of a test plant species. We generally used plants that were 2-4 months old that were grown from seed. However, many of the *Saussurea americana* plants were transplanted from the field, because of scarcity of seed. Cuttings of *Hemizonia minthornii* were used instead of potted plants. After removing the insect from the test plant, the exposed leaf was labeled and preliminary observations were made on the number of eggs oviposited. After 12-21 days, the leaf was removed and inspected under a microscope to confirm counts of eggs and adult feeding holes and to detect egg hatch and larval tunneling. This was sufficiently late enough to allow any eggs

to hatch and larvae to tunnel into the plant. Six weeks after exposure to oviposition, the plants were sealed in screen bags to retain emerging adults. The plants were held until the insects could complete development (up to 3 months), then the plants were dissected to observe signs of insect development and damage. Any plants that deteriorated prematurely were dissected immediately. Loss of such plants sometimes reduced the number of replicates in which developmental data could be collected. In general, we tested 8 replicates per plant species in the *Cardueae* and 4 in the more distantly related taxa. We doubled the number of replicates if there were any signs of larval development.

2) laboratory choice oviposition.

Individual mated females that had completed reproductive diapause were held in a sealed plastic tube with a cut leaf of YST until they oviposited. A female was then placed inside a wooden sleevebox (73 x 43 x 43 cm; length, width, height) containing cut leaves of 4-5 species of test plants for 5 days (Fig. 16). The position of the different species was randomly assigned, and rotated in subsequent trials to avoid any "location" effect. Each species was represented by a clump of two cut leaves held in a vial of water with a foam stopper. A small crumpled paper towel provided a hiding place for the insect. Yellow starthistle leaves were included as a positive control in each trial. The number of adult feeding holes and eggs on each leaf were recorded. We discarded results of any trials in which fewer than 3 eggs were oviposited because they were not considered to pose a sufficient ovipositional challenge to the nontarget plants. The number of useable replicates ranged from 5 to 13 for each of 9 safflower varieties and from 4 to 18 for each of 8 other nontarget species tested. We tested all the native and commercially important species that supported larval development in the no-choice experiments (Fig. 19, Table 6), as well as *Ci. loncholepis*, which was the *Cirsium* that had the highest adult feeding damage. Because safflower is a commercial crop with many varieties, we tested 9 varieties to represent the diversity of this plant.

3) Safflower field attack rate.

Experiments were conducted in eastern Turkey to determine if *C. basicorne* would attack safflower plants at locations where the insect was naturally abundant. The experiments were conducted during 3 years (2002-2004) at 3 sites near Erzurum, Turkey by cooperators from Ataturk University (Erzurum, Turkey) and BBKA (Biotechnology and Biological Control Agency, Rome, Italy) (Fig. 17):

Askale - (39° 58.712' N, 40° 33.783' E, 1580 m elevation) an abandoned cultivated field in alluvial soil near a stream.

Horasan - (40° 07.543, N; 042° 29.941, E; elevation 1501 m) rocky south-facing slope below cliffs beside a stream.

Çat - (39° 34.929' N, 40° 54.210' E, 1814 m elevation) rocky field near the top of a ridge.

Yellow starthistle was naturally present at all sites, and over 80% of YST plants at each of the sites were infested by *Ceratapion* when sampled in 2001. At each site a wire mesh fence was built to protect the experimental plot (about 6 x 12 m) from disturbance by livestock. Each site was visited every other week starting in early April to monitor the presence of adult *C. basicorne* to help determine when to transfer test plants to the field. We tested two accessions of *Centaurea solstitialis*: "US" (seed collected in Davis, California) and "TK" (seed collected at the three sites: Horasan, Çat and Askale), and two commercial safflower (*Carthamus tinctorius*) varieties: CW1221 (linoleic, CalWest) and S317 (oleic, SeedTec). Test plants were grown

indoors from seed before transplanting to the field because of the cold winter conditions in this part of Turkey. The plants were hardened before transfer to the field by moving them to an unheated greenhouse for 5 days (10-23°C), then opening it during the day for 2 days, then opening it day and night for 7 days. Yellow starthistle plants were about 11 weeks old (rosettes with ≥ 4 leaves) and safflower 7 weeks old (10-15 cm tall) when transplanted in the field. Test plants were transplanted in the field on Apr. 24-27, 2002. Plants were placed in small holes dug with minimal disturbance to existing vegetation. We placed 10 plants 50 cm apart in rows spaced 100 cm apart. Each row contained one species, and the rows of species alternated in a regular pattern (YST(US), safflower-oleic, YST(TK), safflower-linoleic), repeated 3 times. Any other naturally occurring YST plants within the plots were removed, but those outside were left undisturbed.

We monitored the phenological development of *C. basicorne* in wild YST plants in the field every week. When the first pupae were observed, we harvested all the test plants. Test plants were harvested on June 1, June 24 and July 5 at Horasan, Çat and Askale, respectively. The leaves and upper stems were removed and the remaining root and lower stem were placed in a zip-lock plastic bag that had a screen panel for ventilation. The bags were held at room temperature (20-25°C) and examined weekly to collect emerging adults, which were pinned and labeled.

We repeated the experiment in 2003 at the Çat and Askale sites. Plants were transferred to the field on May 3-4, respectively. Plants were arranged in alternating rows of 10, as in 2002, with a total of 30 replicates per plant type at Çat, and 30 YST(TR) and 60 oleic at Askale. More oleic replicates were placed at Askale because *Ceratapion* spp. attack of safflower had previously been observed at this site, and oleic was hardier than linoleic. All four plant types (YST(TR), YST(US), linoleic and oleic) were tested at Çat, but only YST(TR) and oleic at Askale. Plants were spaced 40 cm within each row and 80 cm between rows, and were planted within the existing vegetation. Plants were watered at least weekly, as needed. Plants that died were replaced during May 7-13 (21 at Çat, 25 at Askale). Yellow starthistle plants outside the experimental plots were monitored every 7-10 d for the occurrence of *C. basicorne* adult feeding holes, eggs and adults starting on 9 May, 2003.

In 2004, the experiment was repeated at Horasan and Askale sites, using much more safflower (Seedtec-317, oleic; 250 replicates) and only Turkish yellow starthistle (seed from test sites; 40 replicates) as a positive control. Safflower plants were arranged in a plot of 5 by 50 rows, surrounded by YST in 2 rows of 20 along each of the longest sides of safflower plot. We sampled 40 wild YST plants weekly, starting on April 14, to monitor both plant development and *C. basicorne* abundance and phenology. Plants were harvested on June 1 and 18 at Horasan and Askale, respectively.

All adult insects were identified by either Dr. Enzo Colonnelli (University of Rome "La Sapienza", Italy) or Dr. Boris Korotyaev (Russian Academy of Sciences, St. Petersburg).

4.3 Positive Controls

1) laboratory no-choice oviposition and larval development.

Each female was held in a tube with a yellow starthistle leaf until she oviposited before being used on nontarget test plants. After exposure to a nontarget test plant, each female was held in a tube with a YST leaf for up to 3 days to confirm that she was still able to oviposit. If the female did not oviposit on YST, nor on the previous nontarget plant, then the results of the trial were discarded and the plant was retested using a different insect. Larval development was tested by

allowing females to oviposit on YST plants, which were held at the same conditions as for the nontarget test plants.

2) *laboratory choice oviposition.*

Cut leaves of YST were used as one of the plant species in every replicate of the experiment.

3) *field attack rate.*

In each of the experiments, YST plants that had been grown from seed, were transplanted into the field at the same time as the safflower plants and were later harvested and dissected at the same time as the safflower plants.

4.4 Reason for Decisions

1) *laboratory no-choice oviposition and larval development.*

Because females naturally oviposit on the leaves, it was simplest to test plants by confining a female on the leaf of a test plant. Females naturally produce at least 1 egg per day on YST, so a 5-day test period was considered to be more than adequate to determine the ability of this insect to oviposit on a plant. Egg development is 10 days at room temperature, so checking after this interval was suitable for determining the ability of the larva to hatch and tunnel into the leaf petiole. Test plants were then held long enough for larvae to complete development before being dissected.

2) *laboratory choice oviposition.*

Duration of the experiment was chosen to be long enough to allow about 10 eggs to be produced (based on expected oviposition rates), but short enough so that adult feeding would not significantly damage the YST leaves, thus changing the conditions of the experiment. Several leaves of the same species were clumped together to better simulate the appearance of a plant. Potted plants were not used because of the risk of losing insects, which were relatively scarce, and because of the difficulty of finding eggs on so many leaves.

3) *field attack rate.*

Exposure of test plants was designed to occur during the natural period of oviposition of *C. basicorne* in the field. The environmental conditions at the field sites during this season were too cold to grow safflower in the field, because it is not frost tolerant. Therefore, we grew all test plants under artificial conditions in order to produce plants sufficiently large to be attractive to the insect. The plants were transplanted to the field as soon as we saw evidence of oviposition on wild YST plants. The experiments were stopped soon after we saw *C. basicorne* pupae, to prevent insects from emerging before we could collect them. We tried to hold the cut plants under conditions that were moist enough to prevent them from becoming too hard for the insects to emerge, dry enough to discourage microbial growth, and warm enough for the insects to develop. This was a difficult balance to achieve, and the rotting of plants prevented the emergence of many insects.

4.5 Design of Impact Experiment

We grew Yellow starthistle plants from seed in 8-in flower pots in soil mixture containing Supersoil[®], vermiculite and sand (3:1:1 by volume). All plants were grown outdoors and fertilized weekly with Miracle Grow[®] until the start of the experiment on February 4, 2005. Inside quarantine, we infested twenty 6-week old yellow starthistle plants by confining individual females for 2-3 days on intact leaves to oviposit. Two days after the end of

oviposition, the intact leaves were examined under a dissecting microscope to count the number of eggs. We destroyed excess eggs by lancing to achieve infestation rates of either 6 eggs ("low" infestation) and 12 eggs ("high" infestation) per plant. Ten uninfested plants were used as controls. Plants were then moved to a quarantine greenhouse, grown under natural light, and watered as needed (Fig. 18). At week six, each plant was sealed inside a chiffon screen bag to prevent the escape of emerging weevils. At week 10 the plants were measured and dissected, recording plant height and number of weevil larvae, pupae and adults. Aerial plant parts and roots were separated, dried at 70°C for 72-96 h and weighed.

5. Results and Discussion

5.1 Host specificity

1) laboratory no-choice oviposition and larval development.

We tested a total of 51 species of host plants from the Asteraceae family, including 24 native species and 4 economic species (Appendix 2). This includes species from all 5 tribes in the subfamily Cichorioideae and 7 of 8 tribes in the subfamily Asteroideae that contain native North American species or economic species (Appendix 1).¹ In no-choice oviposition tests, *C. basicorne* females oviposited on 94% of plant species in the subtribe Centaureinae, including *Carthamus tinctorius* (safflower) and the native species *Centaurea americana* and *Ce. rothrockii* (Table 6, Fig. 19). Eggs were deposited on 62% of other plant species in the subtribe Carduinae, and most frequently on *Saussurea americana* and *Cirsium loncholepis*. Eggs were observed on only two plants outside the tribe Cardueae: one egg on one plant of *Liatris punctata* and 2 eggs on one plant of *Gazania rigens*, and none of them hatched. These results indicate no risk of larval damage to plants outside the tribe Cardueae.

The highest rates of insect larval development were observed on *Ce. solstitialis* and *Ce. cyanus*, but there was significant development on *Ce. melitensis* (tocalote), *Cnicus benedictus* (blessed thistle), *Carthamus tinctorius* (safflower), and *Crupina vulgaris* (common crupina) (Table 6, Fig. 19). Immature insects possibly developed on only one plant species outside the subtribe Centaureinae. Signs of insect larval damage and an exit hole were observed in roots of two *Saussurea americana* (American basketflower) plants (of 23 replicates). In each case, no adult *C. basicorne* nor any sign of a pupal chamber (which is typical for *C. basicorne*) was found. Therefore, this damage was probably caused by another species of insect. In each case, the plant had been transplanted from the field just before being tested and probably had been infested before entering quarantine. Similar root damage was observed in other *S. americana* plants which were known to not be infested by *C. basicorne*. Thus, it appears that *C. basicorne* cannot develop on this plant and is only able to develop on plants in the subtribe Centaureinae.

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. Such larvae develop in the woody outer portion of the stem, rather than in the central pith. 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 the no-choice experiment, so both these plants require additional choice testing to determine risk of infestation under natural conditions.

¹ *Pluchea odorata*, representing the eighth tribe (Plucheeae), is currently being tested.

There was no larval development in any *Cirsium* species, or in any other threatened or endangered species, or their surrogates, that we tested (Appendix 3). Regarding the two North American native *Centaurea* species, no development was observed in 18 trials of *Ce. rothrockii*. Dead larvae were found in roots of two out of 21 *Ce. americana* trials, but no insects reached the pupal stage. This indicates that there is zero to low risk to North American native plant species.

Intensity of adult feeding on leaves was highly correlated to the number of eggs oviposited in test plants ($R^2 = 0.88$, Fig. 20), 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* (diffuse knapweed) (Fig. 21, Table 6). There was moderate acceptance of eight 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* (*Gazania rigens*). There was at most only trace feeding on test plants in the subfamily *Asteroideae*. These results suggest that under extreme conditions *C. basicorne* 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*.

2) laboratory choice oviposition.

In the sleevebox choice experiment, adult feeding and oviposition by *C. basicorne* was significantly greater on yellow starthistle than on any of the eight other nontarget species tested (Fig. 22). About 74% of eggs were deposited on YST, 20% on *Ce. cyanus* (bachelor's button), 5% on *Ce. melitensis*, 3% on *Sa. americana* and 1% on safflower. These results indicate that *C. basicorne* females are more attracted to YST than to bachelor's button or any of these other nontarget test plants. However, bachelor's button appears to be at risk of some attack, at least under these confined laboratory conditions. A subset of the sleevebox choice trials that tested safflower varieties against YST indicates a low risk of oviposition to 5 of 9 varieties tested (Fig. 23). These experiments were conducted under confined artificial conditions, and in the field the insect is likely to be much more selective (Clement and Cristofaro 1995, Sheppard 1999). Because safflower is a commercial crop field testing was done (see below) to determine if this plant is at risk under natural conditions. Very low attack rates on *Sa. americana* and *Ce. melitensis* indicates a small risk to these plants of adult feeding and oviposition. However, larvae did not develop on *Sa. americana* in the no-choice experiments, so any possible damage to this plant should be very limited. Larvae can develop on *Ce. melitensis* (tocalote, Napa thistle), but this plant is a noxious weed, and such attack is welcome. There was no attack on *Ci. loncholepis*, *Ce. rothrockii* or *Ce. sulphurea*, so these plants are not likely to be at risk in the field.

3) safflower field attack rate.

In 2002, the first signs of adult feeding on foliage of wild YST plants were on 12 Apr., 18 Apr. and 29 Apr. at Çat, Horasan and Askale, respectively (Smith *et al.* 2006, Appendix 6). Infestation of the YST test plants was between 48 and 92% (California and Turkish plants combined) at the three sites, indicating that there was a substantial infestation rate to challenge the safflower plants (Table 7). We reared 61, 101, and 133 individuals of *Ceratapion* from YST at Horasan, Çat and Askale, respectively, but some were immature and others were in poor condition for taxonomic determination. All the adults that could be identified were *C. basicorne* (5, 15, and 15 from Horasan, Çat and Askale, respectively). Infestation of Turkish YST was

higher than that of Californian YST, based on proportion of plants infested, at the three sites. No safflower plants were infested by internal feeding insects at either Horasan or Çat. Seven oleic and five linoleic safflower plants were infested at Askale; however, none of the reared insects were *C. basicorne*. We reared only *C. scalptum* (Mulsant and Rey), *C. orientale* (Gerstaecker), and *C. onopordi* (Kirby) from these safflower plants.

In 2003, signs of *C. basicorne* adult feeding were first observed on April 15 and 20 at Çat and Askale, respectively (Smith *et al.* 2006). By May 9-11, eggs were seen at both locations on nearby wild YST plants. Pupae appeared on June 13-15, and test plants were collected on June 16 and 19 at Çat and Askale, respectively. Repeated sampling of wild YST plants showed that oviposition and larval development occurred during the exposure period of the test plants and that natural infestation rates were very high (80% and 100% at Askale and Çat, respectively). Infestation of YST test plants by *Ceratapion* ranged from 37 to 76% (Table 8), which was lower than that observed on wild YST plants. No safflower plants were infested at Çat, and 3 plants were infested at Askale. Because many plants began to rot while being held for adults to emerge, many of the adult insects could not be identified, including the 3 reared from safflower. All the identifiable insects from YST were *C. basicorne*.

In 2004, signs of *C. basicorne* oviposition were first observed on April 14 and 15 at Askale and Horasan, respectively (Smith *et al.* 2006). *Ceratapion* pupae were first observed on May 27 and June 2, and test plants were collected on June 1 and 18 at Horasan and Askale, respectively. Yellow starthistle rosette diameter increased during the course of the experiment. Yellow starthistle plants at Horasan began to bolt in early May while those at Askale did not bolt before the last observation on 2 June, reflecting the warmer climate at the former location. Eggs of *C. basicorne* were not seen after April 22 at Horasan whereas they were seen until May 28 at Askale. Test plants appear to have been exposed during the peak oviposition period at Askale, but were at the end of the oviposition period at Horasan. Nevertheless, *Ceratapion* infestation of YST test plants was very high at both sites (98 to 100%; Table 9). No safflower plants were infested at Horasan, and 34 plants were infested at Askale. Yellow starthistle was infested primarily by *C. basicorne* (91 adults identified), although a few *C. orientale* were also reared out. Safflower was infested by *C. scalptum* and *C. orientale*, but not by *C. basicorne*.

During three years of field studies in eastern Turkey, we never reared *C. basicorne* from safflower. Deterioration of harvested plants before insects could complete development to provide identifiable specimens was a persistent problem that left some uncertainty about whether any of the unidentifiable insects from safflower could have been *C. basicorne* (Table 10). However, at Çat and Horasan, where *C. basicorne* was the only species present, safflower was never infested by any internal feeding insect, despite infestation rates of 48-98% of the YST test plants and up to 100% infestation of wild YST plants. This indicates a risk of infestation less than 0.27% (1 of 365 safflower plants sampled). At Askale, where *C. scalptum*, *C. orientale* and *C. onopordi* were present, 8 to 26% of safflower plants were infested, but of 19 identifiable insects reared from safflower during 3 years, none were *C. basicorne*.

Our experiments conclusively show that under field conditions *C. basicorne* does not attack safflower, even when 100% of nearby YST is infested. With respect to this crop, the insect is safe to introduce to North America.

5.2 Impact on target plant

In the quarantine laboratory experiment, infestation by weevil larvae caused up to a 23% reduction in size of well-watered, fertilized potted plants grown in a greenhouse (Fig. 24; $F_{(2, 57)}$)

= 3.39, $P < 0.04$). The mean number of insects per plant was 3.1 (± 1.5 sd) in the low infestation treatment and 4.2 (± 3.0) in the high (which is less than was planned). In the field in Turkey, up to seven larvae have been found in YST roots (Uygur *et al.* 2005). So, if this insect attains high densities typical for a successfully established biological control agent, it is likely to cause more damage than we 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% lower seed fertility than uninfested plants (Uygur *et al.* 2005). Although it is difficult to predict how much impact *C. basicorne* will have on YST populations in North America, it is clear 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, which do not feed on these plant parts.

5.3 Summary of Results

No-choice results indicate that no plant species outside the subtribe Centaureinae are at risk of larval damage. 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 oviposition on *Sa. americana*. This plant prefers damp cool habitats and generally does not occur near heavy infestations of YST. 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 laboratory choice and no-choice experiments indicate that *Ce. cyanus* (bachelor's button) is at some 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 (Table 5), but the frequency of such attack has not been studied. 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 oviposition rate on some varieties of this plant under confined conditions. However, during three years of field studies in eastern Turkey, we did not rear any specimens of *C. basicorne* from safflower. At two sites (Çat and Horasan), where *C. basicorne* was the only apionid species present, safflower was not infested by any internal feeding insect, despite infestation rates of 48-98% in YST. At Askale, where *C. scalptum*, *C. orientale* and *C. onopordi* were present, 8 to 34% of safflower plants were infested, but of 19 identifiable insects reared from safflower during 3 years, none were *C. basicorne*.

Clement *et al.* (1989) questioned whether *C. basicorne* was sufficiently host specific to use for classical biological control. However, they were able to conduct only a limited study using a small number of field-collected adults for no-choice oviposition and larval transfer experiments. Use of larval transfer experiments is not very realistic for a species that inserts its eggs into the host plant, because it is normally the adult, rather than the larvae that is able to choose the host plant (Sheppard 1999). Nevertheless, our results confirm Clement *et al.*'s observation that

C. basicorne is physiologically capable of developing on safflower. Clement *et al.* did not conduct any choice experiments, which are crucial to understanding behavior and specificity of the insect in the field (*e.g.*, Clement and Cristofaro 1995, Briese 2005). Their concern that *C. basicorne* might not be safe has been allayed by the laboratory choice and field experiments that we conducted.

Ceratapion basicorne has been reared in the field from only *Ce. solstitialis* (YST), *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 the 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). Our experiments conclusively show that 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.

5.4 Protocol for Releasing the Agent

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, J. Balciunas unpubl. data). Well illustrated keys for these species are available (Wanat 1994, B. A. Korotyaev unpubl. data, Appendix 4). *Ceratapion scalptum*, which attacks safflower, has never been reared from YST, so the risk of accidentally introducing this species can be minimized by using only insects reared from YST. We plan to release individuals that are descendents from the colony in the USDA-ARS quarantine laboratory that was used to conduct our experiments. This population was established by rearing adults from YST 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 hibernial diapause will be released inside large cages (3 x 3 x 2 m) in the spring in dense patches of YST 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 YST 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 U. S. (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.

5.5 Post-Release Monitoring

At the release sites, we 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 YST 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.

5.6 Benefit/Risk

The potential benefit depends upon the degree to which the weevil will reduce the size, survival, and abundance of yellow starthistle. This will not be known until after the weevil is released, because of the influence of environmental effects (YST densities, climate, habitat, lack of specialized natural enemies of the weevil that occur in Eurasia, etc.) that differ from those in the native range of the insect. However, experimental studies suggest that the weevil will reduce plant size, production of viable seed, and possibly survival of immature plants. The weevil has a geographic distribution in Eurasia that matches that of YST, and it often attacks a very high proportion of plants (60-100%). The impact of the weevil should complement those of the established flowerhead insects, the rust pathogen, and interspecific plant competition for light, water, and nutrients (Willis *et al.* 1998, DiTomaso *et al.* 2003a).

Successful biological control will decrease the application of herbicides in rangeland, roadsides and rights-of-way. For example, the California Department of Transportation currently spends \$11.8 million annually on herbicides to control roadside weeds, and YST is a principal weed (R. Melendez pers. comm.). Reduction of YST biomass by biological control will increase soil moisture content in heavily infested areas (Enloe *et al.* 2004, Gerlach 2004), which will benefit production of desirable forage and native plant species and increase water available for human use. The economic benefit of successful biological control of yellow starthistle has been estimated to be between \$40 million and \$1.412 billion, depending on assumptions, in California alone (Jetter *et al.* 2003). Benefits would also occur in Oregon, Washington, Idaho and other states susceptible to invasion by this weed. Possible impact of the weevil on the nontarget species, *Centaurea melitensis*, *Crupina vulgaris* and *Cnicus benedictus*, which are weedy aliens, could also be beneficial.

Releasing this weevil poses no significant risk of damage to any native North American plants, nor to any crops, including those in the sunflower family such as safflower, artichoke, lettuce and sunflower. Although larvae were able to develop on safflower under no-choice laboratory conditions, we never observed larval damage during three years of field experiments conducted at three sites in Turkey in the presence of very high natural attack rates on adjacent YST plants (Smith *et al.* 2006). There is a possibility that the insect may attack bachelor's button (*Centaurea cyanus*). This plant is both a commercial ornamental and an invasive alien weed in the western and southern U.S. (Southeast Exotic Pest Plant Council 1996, Southern Weed Science Society 1998, Whitson *et al.* 2000). The weevil has not been reported as a pest of

bachelor's button in Eurasia, in the native range of the insect. However, it is possible that if the weevil attains high populations on YST in North America, there could be some attack on bachelor's button. Damage to bachelor's button would be in form of small 1-mm² pin holes in the leaves caused by adult feeding and small lumps on the stem near the base of leaves caused by galling reaction to larval feeding. Because the weevil prefers to lay eggs on YST more than on bachelor's button in laboratory experiments, we expect any attack on bachelor's button to be low and transitory (*e.g.*, occurring when the insect population peaks and the YST population begins to decline). When successful biological control ultimately reduces the YST population, the weevil population will also decline, reducing the likelihood of attack on bachelor's button.

6. Potential Environmental Impacts

6.1 Human Impacts

Successful biological control will reduce human exposure to herbicides applied to kill yellow starthistle on rangeland, roadsides, rights-of-way and vineyards. It will reduce physical injury caused by sharp spines on the plant. Humans are not expected to have any contact with the weevil (*Ceratapion basicorne*). Larvae develop inside the plant, and adults are very small and hide most of the year in the bark of trees and possibly in other secluded sites. Adults are active in the early spring, and feed only on yellow starthistle and other closely related thistles and knapweeds (Centaureinae). Some professional beekeepers use yellow starthistle as a nectar source to maintain their European honey bee (*Apis mellifera*) colonies during the summer in California and to produce honey. Although reduction of yellow starthistle populations would reduce this nectar supply, it should also permit other native flowering species to increase which may provide alternate nectar sources for honey bees.

6.2 Potential Economic Impacts

Successful biological control will reduce the costs to ranchers, vineyards, departments of transportation, and other managers of rights-of-way and rangelands who currently manage the weed, primarily by applying herbicides. It should reduce the incidence of "chewing disease" (nigropallidal encephalomalacia) in horses, which is caused by consumption of YST (Cordy, 1978). It should increase soil moisture reserves in annual grasslands in California (Dudley 2000, Enloe *et al.* 2004). Direct economic impact of controlling yellow starthistle in California alone is estimated to be between \$40 million and \$1.4 billion, depending on assumptions (Jetter *et al.* 2003). Additional benefits would occur in Oregon, Washington, Idaho, and other states likely to be invaded by yellow starthistle. The estimated annual benefit of YST to professional beekeepers is about \$200,000 (Maddox *et al.* 1985, Edwards 1989, Goltz 1999). Even if other flowering plant species do not replace YST as a source of summer nectar and pollen, the potential economic cost to beekeepers is very small in comparison to the potential benefits of controlling the weed.

6.3 Plant Impacts

Target weed

The weevil, *Ceratapion basicorne*, in combination with the established flowerhead insects and YST rust, should reduce the size and abundance of yellow starthistle, which is a common invasive weed in rangelands of California, Oregon, Washington and Idaho. Successful biological control will provide self-perpetuating control of the weed population over extensive

areas with little or no negative nontarget impacts. Biological control is not expected to completely eliminate the weed population, but should reduce it to acceptable levels in most habitats. Such results have been seen after the release of other root-feeding insect biological control agents on other rangeland weeds, including tansy ragwort, leafy spurge and diffuse knapweed (Powell and Myers 1988, McEvoy *et al.* 1991, Anderson *et al.* 2000, Seastedt *et al.* 2003, Smith 2004b). Reducing size and density of yellow starthistle plants will reduce interference with grazing animals, increase production of desirable forage and grassland species, and help increase biodiversity in rangeland habitats under proper vegetation management.

Nontarget plants

We do not expect the weevil to directly harm any plants outside the tribe Cardueae (thistles) in the Asteraceae family. Some adult feeding (1-mm² holes) and oviposition 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. Although the weevil is physiologically capable of developing on safflower under laboratory conditions, we observed no attack by this insect under a variety of conditions during three years of field experiments, and conclude that this crop is not at risk of damage. 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*). This damage is most likely to occur during the transitory period when YST populations decrease in the presence of high densities of weevils. It should be noted that neither tocalote nor common crupina have been reported as host plants of this weevil in Eurasia, so our prediction of nontarget damage is probably an overestimation of the risk. The insect has previously been reported to have been reared from bachelor's button and blessed thistle, so these plants are more likely to be attacked. All four of these plants are weedy aliens; however, bachelor's button is also a cultivated ornamental. Possible damage on bachelor's button would be as small holes on leaves (1 mm²) caused by adult feeding and small bumps (5 mm) in the stem near the base of leaf petioles where larvae may occur. Neither of these would be expected to occur very high up on the plant, and thus would probably not be noticeable in cut flowers for floral display. Risk of larval damage to bachelor's button would only be during April to May, because of the limited oviposition period of this insect.

We tested 51 species of plants in two subfamilies of Asteraceae, representing all four tribes in the subfamily Cichorioideae, 7 of 8 tribes in the subfamily Asteroideae, and all genera in the tribe Cardueae containing species of concern.¹ No other plants have been identified that have similar chemistry and/or superficial plant morphology that might cause them to be at risk of attack by this weevil.

6.4 Nonplant Impacts

Relatively little is known about the interactions of yellow starthistle with native North American vertebrates and invertebrates. No threatened or endangered species are known to benefit directly from the presence of YST. Because YST displaces native rangeland plants, decreasing the weed

¹ *Pluchea odorata*, representing the eighth tribe (Plucheeae), is currently being tested.

population should help increase native plant populations and benefit the native species that depend on them. YST is pollinated by the European honey bee (*Apis mellifera*) (Barthell *et al.* 2000, 2001), which is a benefit to professional beekeepers (Maddox *et al.* 1985, Edwards 1989, Goltz 1999), but which is detrimental to native bee species and to native flowers that depend on pollination by the native bees (Barthell *et al.* 2000). Yellow starthistle produces seed that is consumed by some birds (including ring-necked pheasant, mourning dove, California quail, gold finches and house finches) and rodents (Martin *et al.* 1951, Roché 1965). However, seed produced by YST is at the expense of that produced by native forbs and grasses which are displaced by the weed. In general, native wildlife is expected to thrive better with native plants, with which they evolved, than with introduced plants, and we know of no evidence that shows that YST is more beneficial to native wildlife than native plants. If some wildlife, such as deer, were discovered to graze on YST rosettes, it is similarly likely that the weed would not provide better forage than native plants, especially when the overabundance of the invasive species limits the variety of plants available to wildlife. In any case, when birds feed on YST seed, some viable seed can pass through their guts, which increases the risk of dispersing the weed to new locations (Roché 1965). Larvae of the native painted lady butterfly (*Vanessa cardui*) feed on YST leaves (Pitcairn *et al.* 1999b); however, they also feed on other native species of *Cirsium*, common alien Cardueae species, hollyhock and mallow (Malvaceae), and various legumes (Fabaceae) (Struttmann 2005). The butterfly has a Nature Conservancy Global Rank of G5 (demonstrably secure globally, though it may be quite rare in parts of its range) and is not expected to be threatened by a decrease in YST populations. Thus, in general, successful biological control of yellow starthistle is expected to benefit rather than harm native arthropods and vertebrates.

6.5 Proposed Methods for Mitigation

No undesirable effects are anticipated except for the possible damage to bachelor's button grown for ornamentals. If necessary, weevils may be killed by application of systemic insecticides on infested plants or by destroying their host plant, yellow starthistle, using herbicides, tillage, flaming or hand pulling.

6.6 Abiotic and Edaphic Effects

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). Successful biological control of YST will help reduce the depletion of ground water in heavily infested rangeland in California and Oregon (Borman *et al.* 1992, Dudley 2000, Enloe *et al.* 2004). This will increase the amount of ground water available to desirable rangeland vegetation and for other human uses.

6.7 Outcome of No Action

Yellow starthistle has invaded 20 million acres of the Pacific West and threatens to expand further east. Infested rangeland currently causes reduced production of cattle, interferes with recreational use, depletes soil moisture, and reduces desirable biodiversity. Herbicides are currently being applied along roadsides and rights-of-way, and on intensively managed or high value land. Without action, these control activities will continue, costing millions of dollars annually and exposing people and the environment to herbicides. Any failure of current control methods (such as herbicide resistance) or decrease in control activities will allow the weed to increase densities and expand the area infested.

7. Petitioner's Conclusion

The results of the host specificity experiments conducted in quarantine laboratory and field experiments conducted in Turkey indicate that the weevil, *Ceratapion basicorne*, will not damage or develop on any plant other than a few species in the subtribe Centaureinae, all of which are alien weeds in North America. However, one of these species, bachelor's button (*Centaurea cyanus*), is grown commercially as an ornamental. It is possible that the weevil will cause some damage to the stems of this plant, creating a small bump where the larva develops. However, the weevil has a short season when it is capable of laying eggs (April to May), and it is adapted to attacking rosettes, which bachelor's button does not form. Furthermore, this insect has not been reported to be a pest of ornamental bachelor's button in Eurasia, where this insect is native. Thus, any damage to bachelor's button is expected to be infrequent and minor. The other plants likely to be attacked are the alien weeds: tocalote (*Centaurea melitensis*) common crupina (*Crupina vulgaris*), and blessed thistle (*Cnicus benedictus*).

The weevil is capable of reducing the growth of yellow starthistle (YST) and production of viable seed, and it is likely to reduce plant survival. Because *C. basicorne* feeds on the rootcrown and leaves, it is not likely to directly interfere with the previously released flowerhead insects, which do not feed on these plant parts. We expect the weevil to disperse widely on its own and to permanently reduce the size and the abundance of yellow starthistle. The potential benefits are:

- 1) decreased costs of controlling the weed on roadsides, rights-of-way, and rangeland,
- 2) decreased environmental contamination caused by application of herbicides,
- 3) increased production of forage and native plant species on YST-infested land, and
- 4) protection of land at risk of being invaded by YST.

Possible losses could be incurred by:

- 1) commercial vendors and applicators of herbicides that are currently employed to control YST,
- 2) beekeepers who exploit yellow starthistle nectar and pollen, and
- 3) producers and vendors of ornamental bachelor's button.

The anticipated potential benefits appear to greatly outweigh the anticipated potential losses. We therefore recommend that the weevil be released experimentally to determine if it will reduce the size and abundance of yellow starthistle under field conditions. If it causes becomes established and impacts the target weed (determinable within several years), then it will be recommended for widespread distribution.

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Tables

Table 1. Subspecies of yellow starthistle and their Old World distributions (Wagenitz 1975, Dostál 1976).

Scientific Name	Distribution
<i>Centaurea solstitialis</i> L. subsp. <i>solstitialis</i>	almost throughout range of species
<i>Centaurea solstitialis</i> L. subsp. <i>adamii</i> (Willd.) Nyman	c. and e. Mediterranean region; Crimea.
<i>Centaurea solstitialis</i> L. subsp. <i>carneola</i> (Boiss.) Wagenitz	endemic to e. Mediterranean
<i>Centaurea solstitialis</i> L. subsp. <i>erythracantha</i> (Halácsy) Dostál	e. Greece
<i>Centaurea solstitialis</i> L. subsp. <i>pyracantha</i> (Boiss.) Wagenitz	endemic to e. Mediterranean
<i>Centaurea solstitialis</i> L. subsp. <i>schouwii</i> (DC.) Dostál	endemic to Sicily and Sardinia

Table 2. Some taxonomic relationships among *Centaurea* species that occur in North America.

Species ¹	Common name	Subgenus ¹	Pollen type ²	Section ²
<i>C. calcitrapa</i>	purple starthistle	Calcitrapa	Jacea	<i>Calcitrapa</i> DC.
<i>C. virgata</i> var. <i>squarrosa</i>	squarrose knapweed		Jacea	<i>Acrolophus</i> (Cass.) DC.
<i>C. diffusa</i>	diffuse knapweed	Centaurea	Jacea	<i>Acrolophus</i> (Cass.) DC.
<i>C. maculosa</i> [= <i>biebersteinii</i> , = <i>stoebe</i>]	spotted knapweed	Centaurea	Jacea	<i>Acrolophus</i> (Cass.) DC.
<i>C. cineraria</i>	dusty miller	<i>Acrolophus</i> (Cass.) Dobroc.	Jacea	<i>Pannophyllum</i> Hay.
<i>C. x pratensis</i> (<i>nigra x jacea</i>) [= <i>debauxii</i>]	meadow knapweed	Jacea (Miller) Hayek	Jacea	<i>Eujaceae</i> Boiss.
<i>C. solstitialis</i>	yellow starthistle	<i>Solstitiaria</i> (Hill) Dobroc.	Jacea	<i>Mesocentron</i> (Cass.) DC.
<i>C. melitensis</i>	tocalote, Napa or Malta thistle	<i>Solstitiaria</i> (Hill) Dobroc.	Jacea	<i>Mesocentron</i> (Cass.) DC.
<i>C. sulphurea</i>	Sicilian starthistle	<i>Solstitiaria</i> (Hill) Dobroc.	Jacea	<i>Mesocentron</i> (Cass.) DC.
<i>C. cyanus</i>	batchelor button, cornflower	<i>Cyanus</i> (Miller) Hayek ³	Cyanus	<i>Cyanus</i> DC., <i>Annuae</i> Boiss.
<i>C. montana</i>	mountain bluet	<i>Cyanus</i> (Miller) Hayek ³	Montana	<i>Cyanus</i> DC., <i>Perennes</i> Boiss.
<i>C. dealbata</i>	Persian cornflower	<i>Psephellus</i> (Cass.) Schmalh ³	Dealbata	<i>Psephellus</i> (Cass.) DC.
<i>C. americana</i>	American basketflower		Serratula	<i>Plectocephalus</i> (D. Don) DC.
<i>C. rothrockii</i>	Rothrock's basketflower		Serratula	<i>Plectocephalus</i> (D. Don) DC.

¹ Dostál (1976).² Wagenitz (1975). Note that *Cnicus* was merged into *Centaurea* (Garcia-Jacas *et al.* 2000, Greuter 2003), and it has *Jacea* pollen type (Wagenitz & Hellwig 1994). *Plectocephalus* was segregated as a genus by Garcia-Jacas *et al.* (2001).³ *Psephellus* and *Cyanus* were segregated as genera by Greuter (2003).

Table 3. Geographic distribution of 101 native *Cirsium* species in the contiguous United States. Individual species may occur in more than one region.

Region ^a	No. of species
California	29
Pacific Northwest	23
Inter-Mountain	35
Great Basin	34
Southwest	32
Southern Plains	12
Northern Plains	10
Southeast	11
Mid-Atlantic	10
New England	8
Ohio Valley	11
Great Lakes	10

^aState-groupings by Region: CALIFORNIA: California; PACIFIC NORTHWEST: Oregon, Washington, Idaho; INTER-MOUNTAIN: Montana, Wyoming, Colorado; GREAT BASIN: Nevada, Utah; SOUTHWEST: Arizona, New Mexico; SOUTHERN PLAINS: Texas, Oklahoma, Kansas; NORTHERN PLAINS: Nebraska, South Dakota, North Dakota; SOUTHEAST: Louisiana, Mississippi, Alabama, Georgia, Florida, South Carolina, North Carolina, Tennessee, Missouri; MID-ATLANTIC: Virginia, West Virginia, Maryland, Delaware, New Jersey; NEW ENGLAND: New York, New Hampshire, Vermont, Connecticut, Massachusetts, Rhode Island, Maine; OHIO VALLEY: Iowa, Illinois, Indiana, Ohio, Pennsylvania; GREAT LAKES: Minnesota, Wisconsin, Michigan

Table 4. Biological control agents introduced to North America. All the insects attack the flowerheads, and the rust primarily attacks young plants in the spring.

Biological control agent	Common name ¹	First release	Status
<i>Urophora jaculata</i>		1969	Never established in USA.
<i>Urophora sirunaseva</i>	YST gall fly	1984	Widely established, present at most YST infestations in CA & OR; a few sites in WA, ID.
<i>Bangasternus orientalis</i>	YST bud weevil	1985	Widespread in CA, OR, WA & ID.
<i>Chaetorellia australis</i>	YST peacock fly	1988	Prefers bachelor button; established at a few sites in CA; widespread in OR, WA, ID.
<i>Eustenopus villosus</i>	YST hairy weevil	1990	Well established in CA; widespread in OR, WA; a few sites in ID, UT.
<i>Larinus curtus</i>	YST flower weevil	1992	Established at a few sites in CA, WA, ID; widespread in OR.
<i>Puccinia jacea</i> var. <i>solstitialis</i>	YST rust	2003	Initial releases have been made at 20 sites in CA.
<u>Unapproved accidental introduction:</u>			
<i>Chaetorellia succinea</i> ²	YST false peacock fly	1991	Widely established in CA & OR, and spreading into WA, ID & NV.

¹ YST = yellow starthistle

² This species was never approved for introduction to N. America, but it was evaluated for risk to nontarget plants after its appearance (Balciunas and Villegas 2001, Balciunas unpubl. data).

Table 5. Reported host plants of *Ceratapion basicorne* collected in the field.

Adult reared from	Adult resting on
<i>Centaurea solstitialis</i> L. ^{1,2,3,4}	<i>Centaurea calcitrapa</i> L. ^{1,2,3}
<i>Centaurea cyanus</i> L. ("associated with") ² , flowerheads ⁵	<i>Centaurea jacea</i> L. ^{1,2} <i>Centaurea rhenana</i> Bor. ² (=stoebe, = maculosa, = paniculata)
<i>Centaurea depressa</i> M.Bieb. ⁴	<i>Centaurea scabiosa</i> L. ^{1,2}
<i>Cnicus benedictus</i> L. ⁴	<i>Centaurea virgata</i> Lam. ^{1,2} <i>Arctium lappa</i> L. ^{1,2} <i>Carduus pycnocephalus</i> L. ^{1,2} <i>Carduus tenuiflorus</i> Curtis ^{1,2} <i>Onopordum tauricum</i> Wild. ^{1,2}

¹ Alonso-Zarazaga 1990a² Wanat 1994³ Campobasso *et al.* 1999⁴ J. Balciunas (unpubl. data)⁵ Dieckmann (1977)

Table 6. 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	eggs/day	No. of trials development	Percentage of trials			
					Adult feeding holes present	eggs present	Internal plant damage present	Adults or pupae present
Subfamily Cichorioideae, tribe Cardueae, subtribe Centaureinae								
<i>Acroptilon repens</i> ⁸	10	4.1	0.2	9	90	40	0	0
<i>Carthamus tinctorius</i> ¹	100	10.7	0.5	76	57	61	42	38
<i>Centaurea americana</i> ⁶	23	6.0	0.2	21	91	43	9	0
<i>Centaurea calcitrapa</i> ⁸	8	9.6	0.2	8	100	75	0	0
<i>Centaurea cineraria</i> ⁷	10	0.5	0.0	10	80	0	0	0
<i>Centaurea cyanus</i> ^{7,8}	8	22.7	1.7	8	100	100	100	100
<i>Centaurea dealbata</i> ⁸	11	10.7	0.8	13	100	82	23	0
<i>Centaurea debeauxii</i> (= <i>C. x pratensis</i>) ⁸	16	9.4	0.6	15	94	56	50	13
<i>Centaurea diffusa</i> ⁸	8	24.3	1.5	7	100	88	0	0
<i>Centaurea maculosa</i> ⁸	8	8.8	0.6	6	100	75	0	0
<i>Centaurea melitensis</i> ⁸	20	11.2	0.6	17	95	75	47	47
<i>Centaurea montana</i> ⁷	17	4.3	0.2	15	53	35	13	7
<i>Centaurea rothrockii</i> ⁶	18	12.8	0.3	15	100	56	0	0
<i>Centaurea solstitialis</i> ^{2,8}	350	16.6	1.5	32	100	99	88	88
<i>Centaurea sulphurea</i> ⁸	14	4.9	0.3	12	79	50	17	17
<i>Centaurea virgata</i> var. <i>squarrosa</i> ⁸	16	7.9	0.5	10	100	81	0	0
<i>Cnicus benedictus</i> ⁸	8	5.6	0.5	9	100	88	70	56
<i>Crupina vulgaris</i> ⁸	9	3.5	0.2	11	89	44	36	36
Subfamily Cichorioideae, tribe Cardueae, other subtribes								
<i>Carduus pycnocephalus</i> ⁸	10	1.7	0.2	9	40	10	0	0
<i>Cirsium brevistylum</i> ⁶	7	0.0	0.1	7	0	29	0	0
<i>Cirsium ciliolatum</i> ⁶	9	0.1	0.0	9	22	0	0	0
<i>Cirsium cymosum</i> ⁶	11	0.1	0.0	8	18	0	0	0

Table 6. (Continued)

Plant species	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
<i>Cirsium fontinale</i> ⁶	15	0.1	0.0	15	27	0	0	0
<i>Cirsium hydrophilum</i> var. <i>vaseyi</i> ⁶	16	0.7	0.0	16	56	13	0	0
<i>Cirsium loncholepis</i> ⁶	9	4.0	0.4	9	78	56	0	0
<i>Cirsium occidentale</i> var. <i>venustum</i> ⁶	16	1.4	0.0	15	44	6	0	0
<i>Cirsium vinaceum</i> ⁶	7	0.0	0.0	7	0	0	0	0
<i>Cirsium vulgare</i> ⁸	8	0.4	0.1	8	13	13	0	0
<i>Cynara scolymus</i> ³	11	1.2	0.0	11	55	0	0	0
<i>Echinops exaltatus</i> ⁷	4	1.8	0.05	4	100	25	0	0
<i>Onopordum acanthium</i> ⁸	8	0.8	0.1	8	88	25	0	0
<i>Saussurea americana</i> ⁶	41	3.9	0.3	23	73	51	9	9
<i>Silybum marianum</i> ⁸	8	0.2	0.0	8	38	0	0	0
<i>Xeranthemum cylindraceum</i> ⁷	4	3.4	0.2	4	75	25	0	0
Subfamily Cichorioideae, other tribes								
<i>Gazania rigens</i> ⁷	10	2.2	0.04	10	40	10	0	0
<i>Trixis californica</i> ⁶	5	0.1	0.0	5	40	0	0	0
<i>Agoseris grandiflora</i> ⁶	8	0.0	0.0	8	0	0	0	0
<i>Lactuca sativa</i> ⁴	8	0.0	0.0	8	0	0	0	0
<i>Stephanomeria cichoriacea</i> ⁶	4	0.0	0.0	2	0	0	0	0
<i>Stokesia laevis</i> ⁶	4	0.0	0.0	4	0	0	0	0
Subfamily Asteroideae								
<i>Artemisia californica</i> ⁶	6	.3	0.0	6	17	0	0	0
<i>Symphytotrichum</i> (= <i>Aster</i>) <i>chilense</i> ⁶	5	.0	0.0	5	0	0	0	0
<i>Brickellia californica</i> ⁶	4	.4	0.0	4	75	0	0	0

Table 6. (Continued)

Plant species	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
<i>Liatriis punctata</i> ⁶	5	.7	0.04	4	60	20	0	0
<i>Pseudognaphalium</i> (= <i>Gnaphalium</i>) <i>californicum</i> ⁶	6	.0	0.0	5	0	0	0	0
<i>Eriophyllum</i> <i>stoechadifolium</i> ⁶	7	.4	0.0	7	43	0	0	0
<i>Hemizonia minthornii</i> ⁶	4	.0	0.0	4	0	0	0	0
<i>Echinacea purpurea</i> ⁶	6	.2	0.0	6	17	0	0	0
<i>Helianthus annuus</i> ^{5,6}	4	.0	0.0	4	0	0	0	0
<i>Senecio bicolor</i> ssp. <i>cineraria</i> ⁷	8	.0	0.0	6	0	0	0	0
<i>Senecio vulgaris</i> ⁸	4	.4	0.0	4	25	0	0	0

¹ safflower

² yellow starthistle, target weed

³ artichoke

⁴ lettuce

⁵ sunflower

⁶ native to North America

⁷ alien ornamental

⁸ alien weed

Table 7. Infestation of root crowns or lower stems of test plants by apionid weevils (including larvae, pupae and adults) at three field test sites in Turkey in 2002. *Ceratapion basicorne* was reared only from yellow starthistle.

Site	Test plant ^a			
	Yellow starthistle		Safflower	
	YST(US)	YST(TK)	Oleic	Linoleic
Proportion of plants infested (%) ^b				
Horasan	83.3 b	100.0 a	0.0 c	0.0 c
Çat	27.9 b	66.7 a	0.0 c	0.0 c
Askale	58.6 b	86.7 a	19.0 c ^c	15.8 c ^d
Number of insects per infested plant (\pm SE) ^e				
Horasan	1.4 \pm 0.2 a	1.5 \pm 0.2 a		
Çat	2.3 \pm 0.5 a	2.8 \pm 0.4 a		
Askale	2.6 \pm 0.5 a	2.4 \pm 0.3 a	1.8 \pm 0.8 a	1.0 \pm 0.0 a
Number of plants sampled				
Horasan	24	24	22	23
Çat	26	30	25	13
Askale	29	30	21	19

^a YST(US) = yellow starthistle seed from California, YST(TR) = yellow starthistle seed from Turkey, Oleic = safflower variety SeedTec S317, Linoleic = safflower variety CalWest 1221.

^b Values followed by the same letter in the same row are not significantly different (chi-square test, $P < 0.05$).

^c Adults identified: 4 *C. scalptum*, 1 *C. orientale*, 2 *C. onopordi*.

^d Adults identified: 2 *C. scalptum*.

^e Values followed by the same letter in the same row are not significantly different (Fisher's protected LSD, $\alpha < 0.05$).

Table 8. Infestation of root crowns or lower stems of test plants by apionid weevils (including larvae, pupae and adults) at two field test sites in Turkey in 2003. *Ceratapion basicorne* was reared only from yellow starthistle.

Site	Test plant ^a			
	Yellow starthistle		Safflower	
	YST(US)	YST(TK)	Oleic	Linoleic
Proportion of plants infested (%) ^b				
Çat	37.0 a	44.8 a	0.0 b	0.0 b
Askale	--	76.7 a	7.7 b ^c	--
Number of insects per infested plant (\pm SE) ^d				
Çat	5.0 \pm 0.8 a	3.4 \pm 0.4 a	--	--
Askale	--	4.1 \pm 0.5 a	1.0 \pm 0.0 b	--
Number of plants sampled				
Çat	27	29	27	30
Askale	--	30	39	--

^a YST(US) = yellow starthistle seed from California, YST(TR) = yellow starthistle seed from Turkey, Oleic = safflower (SeedTec S317), Linoleic = safflower (CalWest 1221).

^b Values followed by the same letter in the same row are not significantly different (chi-square test, $P < 0.01$).

^c 3 adults, all unidentifiable.

^d Values followed by the same letter in the same row are not significantly different (Student's t -test, $P < 0.05$).

Table 9. Infestation of root crowns or lower stems of test plants by apionid weevils (including larvae, pupae and adults) at two field test sites in Turkey in 2004. *Ceratapion basicorne* was reared only from yellow starthistle.

Site	Test plant ^a	
	YST(TR)	Safflower
Proportion of plants infested (%) ^b		
Horasan	98 a ^c	0 b
Askale	100 a ^d	34 b ^e
Number of insects per infested plant (\pm SE) ^f		
Horasan	1.7 \pm 0.2 a	0.0 \pm 0 b
Askale	2.4 \pm 0.2 a	1.09 \pm 0.05 b
Number of plants sampled		
Horasan	40	250
Askale	40	99

^a YST(TR) = yellow starthistle seed from Turkey, Safflower = (SeedTec S317, oleic).

^b Values followed by the same letter in the same row are not significantly different (chi-square test, $P < 0.01$).

^c Adults identified: 24 *C. basicorne*, 1 *C. orientale*.

^d Adults identified: 67 *C. basicorne*, 2 *C. orientale*.

^e Adults identified: 8 *C. scalptum*, 2 *C. orientale*.

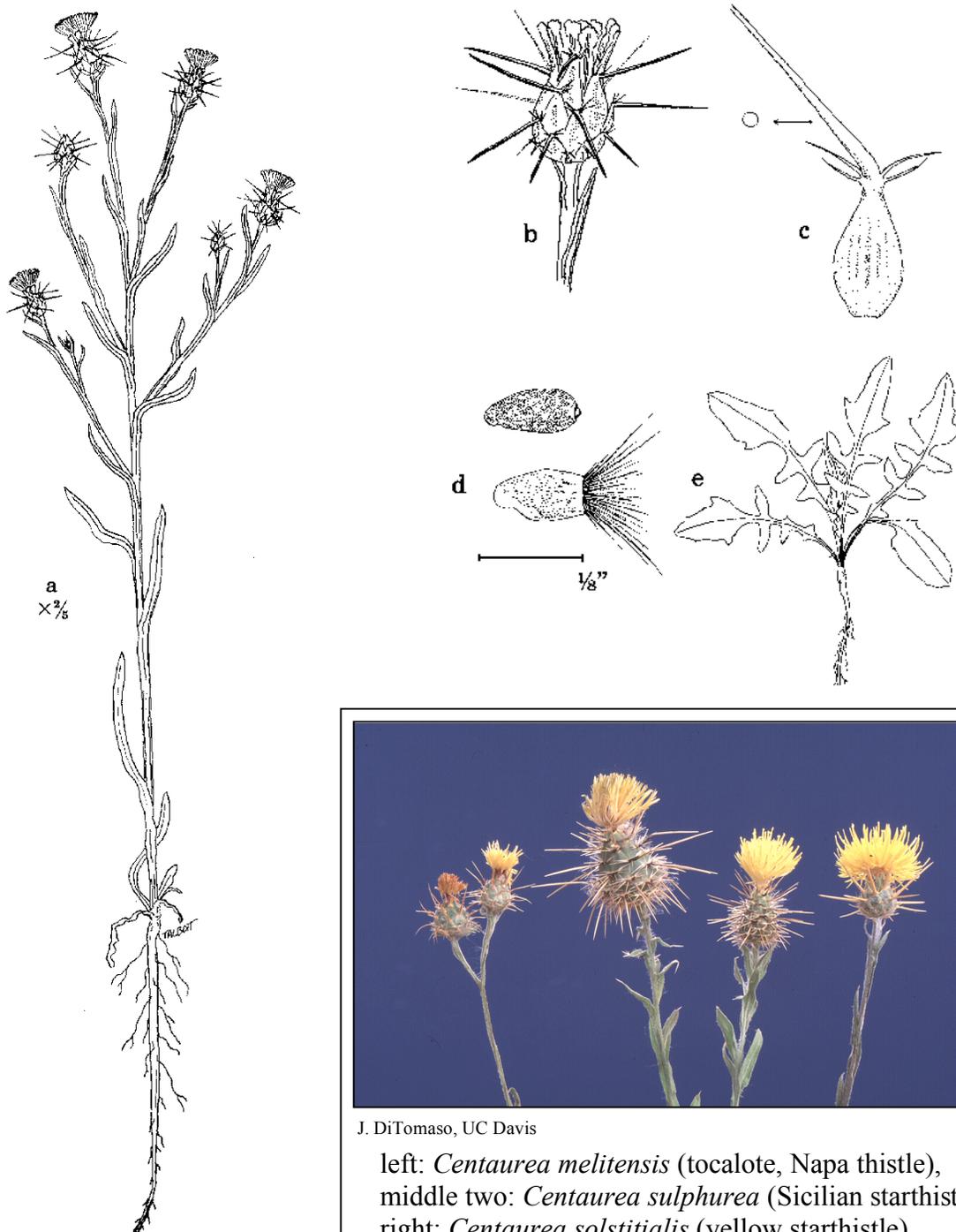
^f Values followed by the same letter in the same row are not significantly different (Student's *t*-test, $P < 0.05$).

Table 10. Identification of apionid weevils infesting root crowns or lower stems of test plants at field sites in Turkey in 2002-2004.

Ceratapion species	Number of individuals ^a					
	Askale		Çat		Horasan	
	YST	Safflower	YST	Safflower	YST	Safflower
2002						
<i>C. basicorne</i>	15	0	15	0	5	0
<i>C. orientale</i>	0	1	0	0	0	0
<i>C. onopordi</i>	0	2	0	0	0	0
<i>C. scalptum caviceps</i>	0	6	0	0	0	0
unidentifiable specimens	118	3	86	0	56	0
2003						
<i>C. basicorne</i>	10	0	15	0	—	—
<i>C. orientale</i>	0	0	0	0	—	—
<i>C. onopordi</i>	0	0	0	0	—	—
<i>C. scalptum caviceps</i>	0	0	0	0	—	—
unidentifiable specimens	61	3	29	0	—	—
2004						
<i>C. basicorne</i>	67	0	—	—	24	0
<i>C. orientale</i>	2	2	—	—	1	0
<i>C. scalptum caviceps</i>	0	8	—	—	0	0
unidentifiable specimens	24	5	—	—	1	0

^a YST = combined results of Turkish and Californian plants, Safflower = combined results of oleic and linoleic varieties.

Figures



J. DiTomaso, UC Davis

left: *Centaurea melitensis* (tocalote, Napa thistle),
 middle two: *Centaurea sulphurea* (Sicilian starthistle),
 right: *Centaurea solstitialis* (yellow starthistle).

Figure 1. Illustrations of Yellow starthistle, *Centaurea solstitialis*, (from Roché and Roché 2000) (a - mature plant, b - flowerhead (capitulum), c - spine on bract (round cross-section), d - seed (achene), e - rosette). Photograph showing differences among the three yellow *Centaurea* species that occur in North America (courtesy J. DiTomaso).

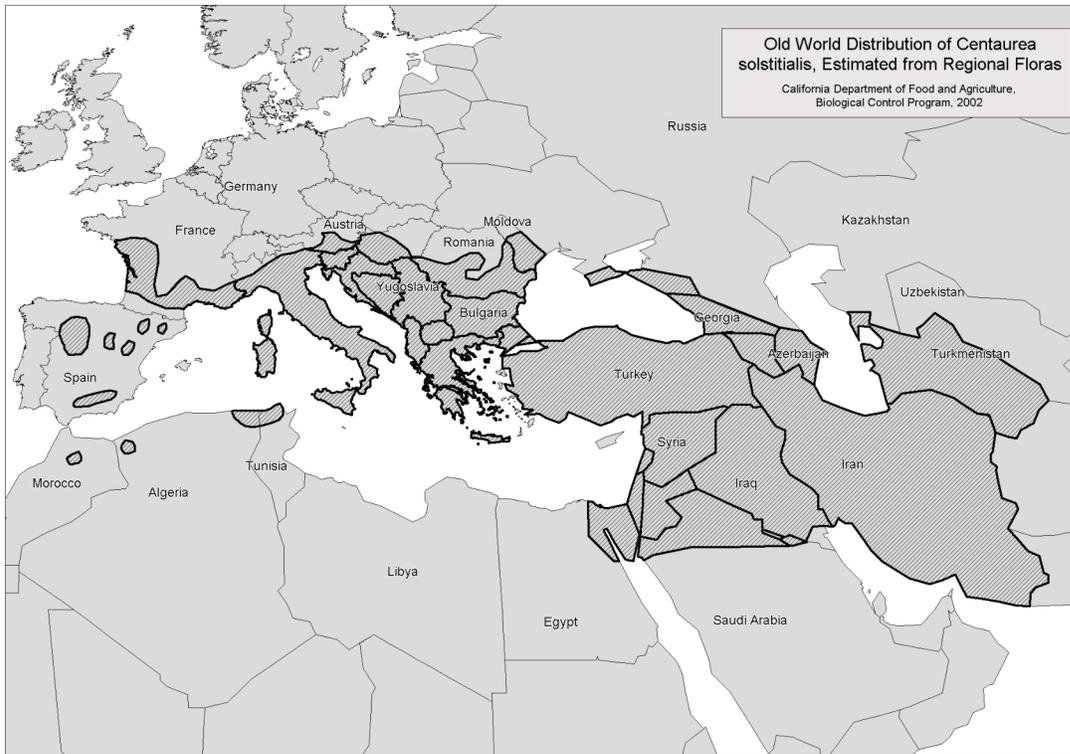


Figure 2. Geographic distribution of yellow starthistle in the Old World based on several Eurasian floras (from CDFA).

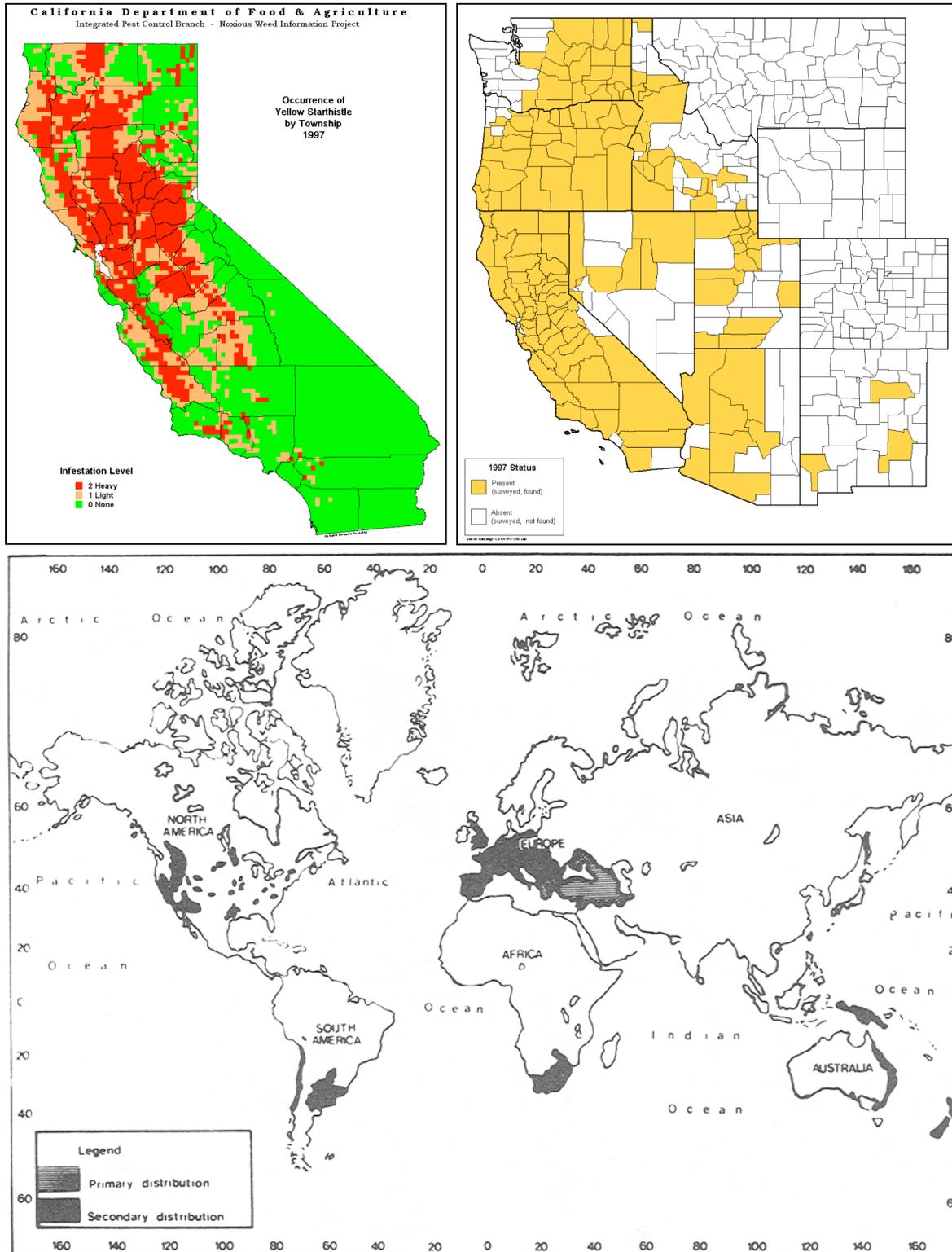


Figure 3. Geographic distribution of yellow starthistle in California (from CDFA), the western United States (from Sheley *et al.* 1999), and the world (from Maddox 1985).

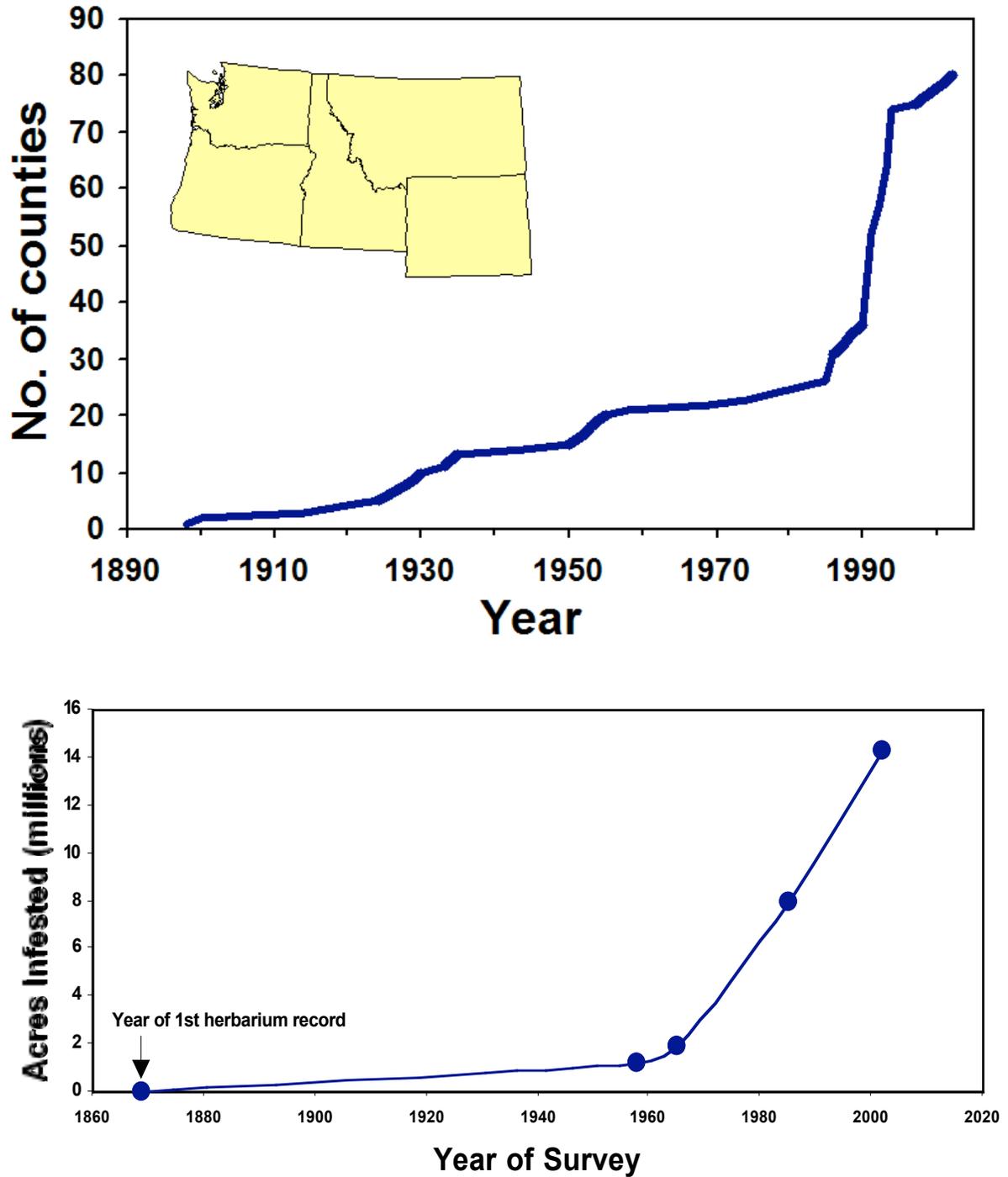


Figure 4. Historical spread of yellow starthistle in the Pacific Northwest (top graph; from Rice 2005) and in California (bottom graph; from Pitcairn *et al.* in press).

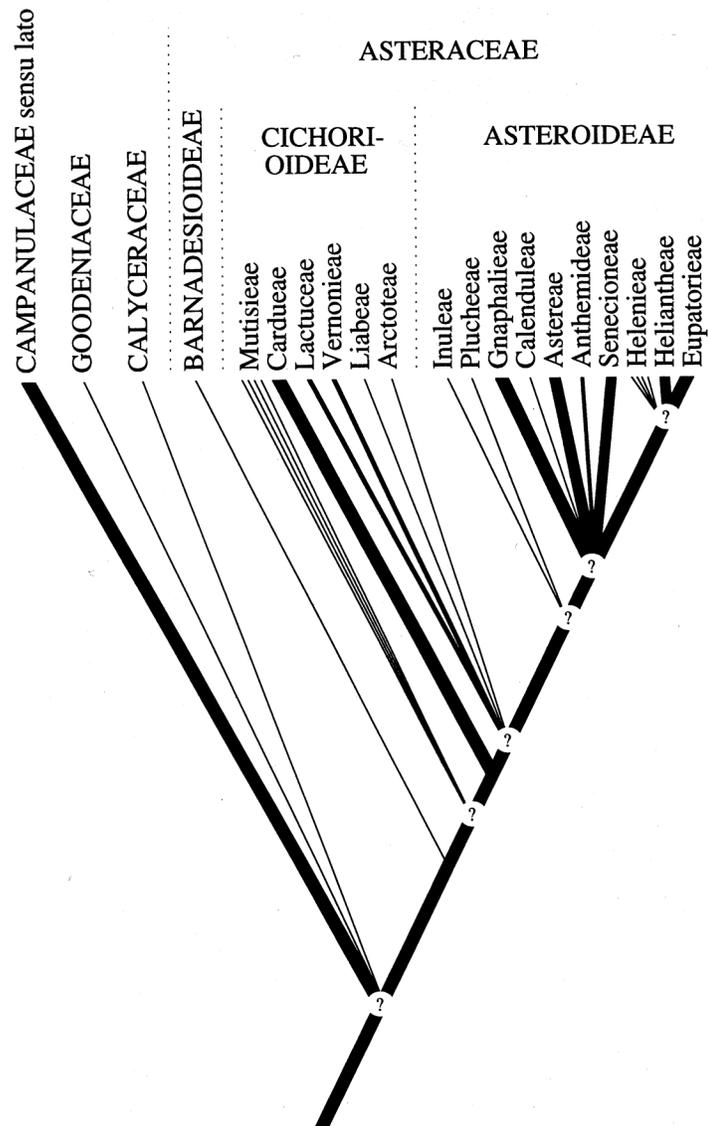


Figure 5. Current state of knowledge of phylogenetic relationships of family Asteraceae (from Bremer 1994). The Asteraceae contains three subfamilies, but only Cichorioideae and Asteroideae occur in North America. Yellow starthistle is in the subfamily Cichorioideae, tribe Cardueae. Asteroideae is monophyletic but Cichorioideae is paraphyletic (*i.e.*, Cichorioideae has no single common ancestor that is not also a common ancestor of Asteroideae).

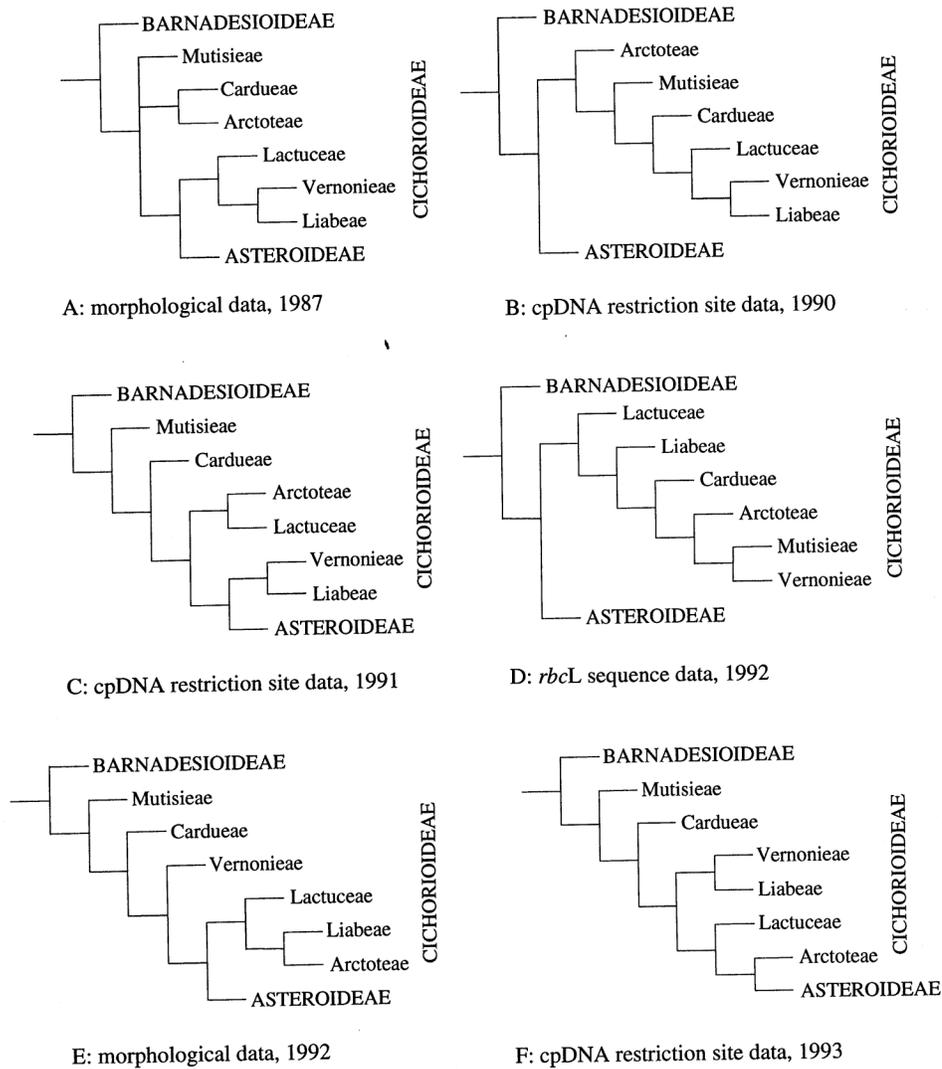


Figure 6. Current data support several different cladistic hypotheses of the taxonomic relationships among tribes of the subfamily Cichorioideae and the subfamily Asteroideae (from Bremer 1994). There is not enough data to determine which (if any) of these is correct.

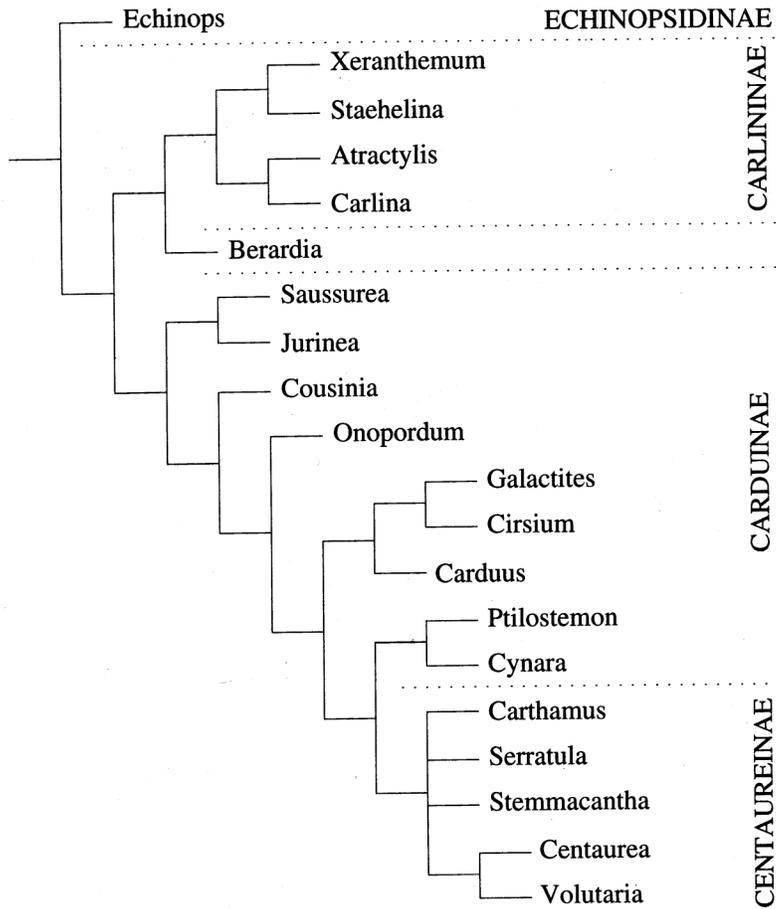


Figure 7: A cladistic hypothesis of relationships within the tribe Cardueae, based on a strict consensus tree of five equally parsimonious cladograms using 23 flower and seed characteristics (from Bremer 1994). Genera that contain species growing in North America are: *Carduus*, *Carthamus*, *Centaurea*, *Cirsium*, *Cynara*, *Onopordum*, *Saussurea*.

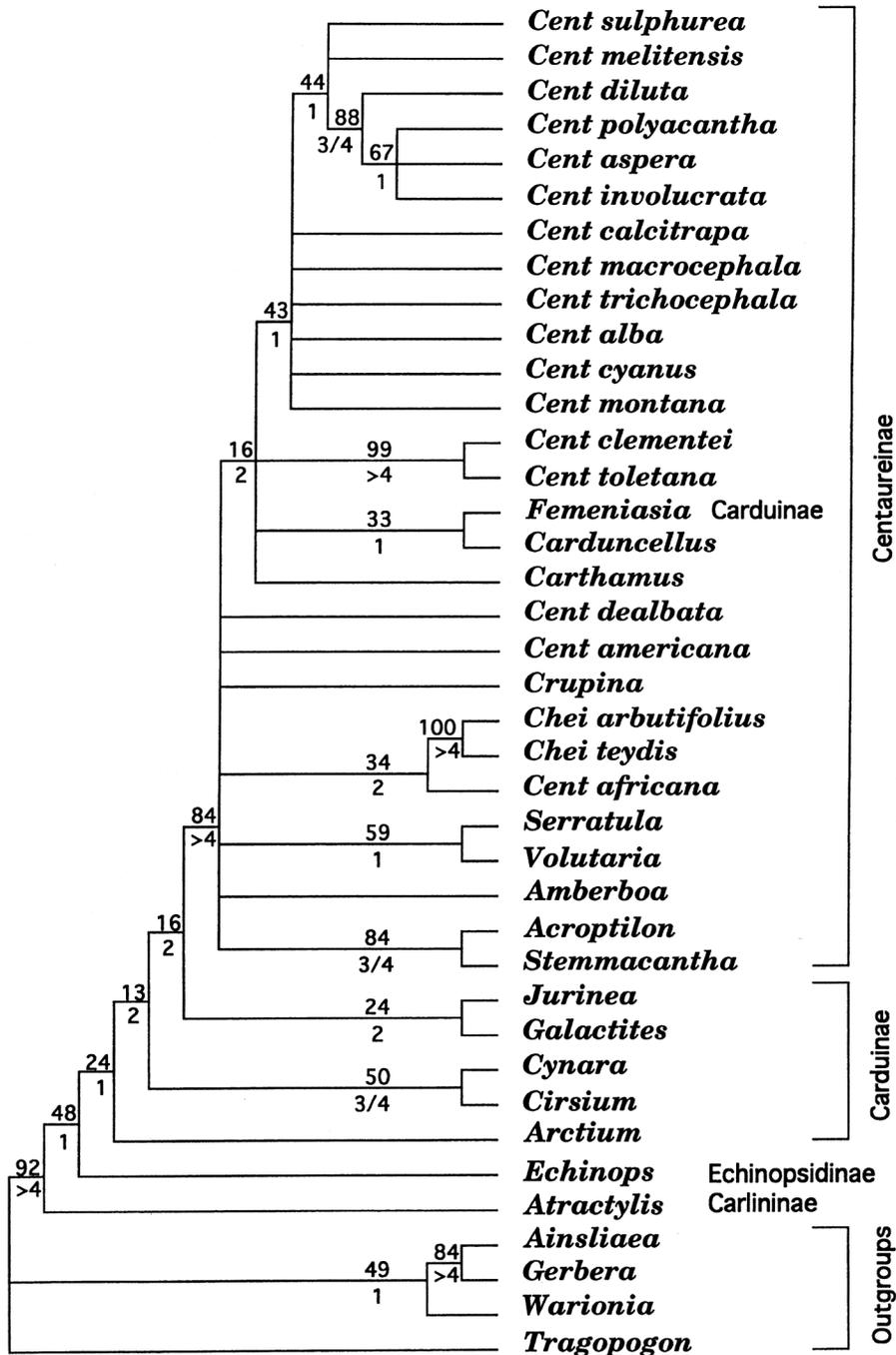


Figure 8. Phylogenetic relationships in tribe Cardueae based on ITS DNA sequences. Subtribal affiliations are shown by brackets and designations to right of generic names. Numbers above horizontal lines are bootstrap values and represent the percentage of trees generated in 100 bootstrap replicates that showed each grouping. Numbers below branches are decay indices and indicate the number of steps longer than the most parsimonious trees at which the branches collapse (from Susanna *et al.* 1995). (*Cent* = *Centaurea*, *Chei* = *Cheirolophus*).

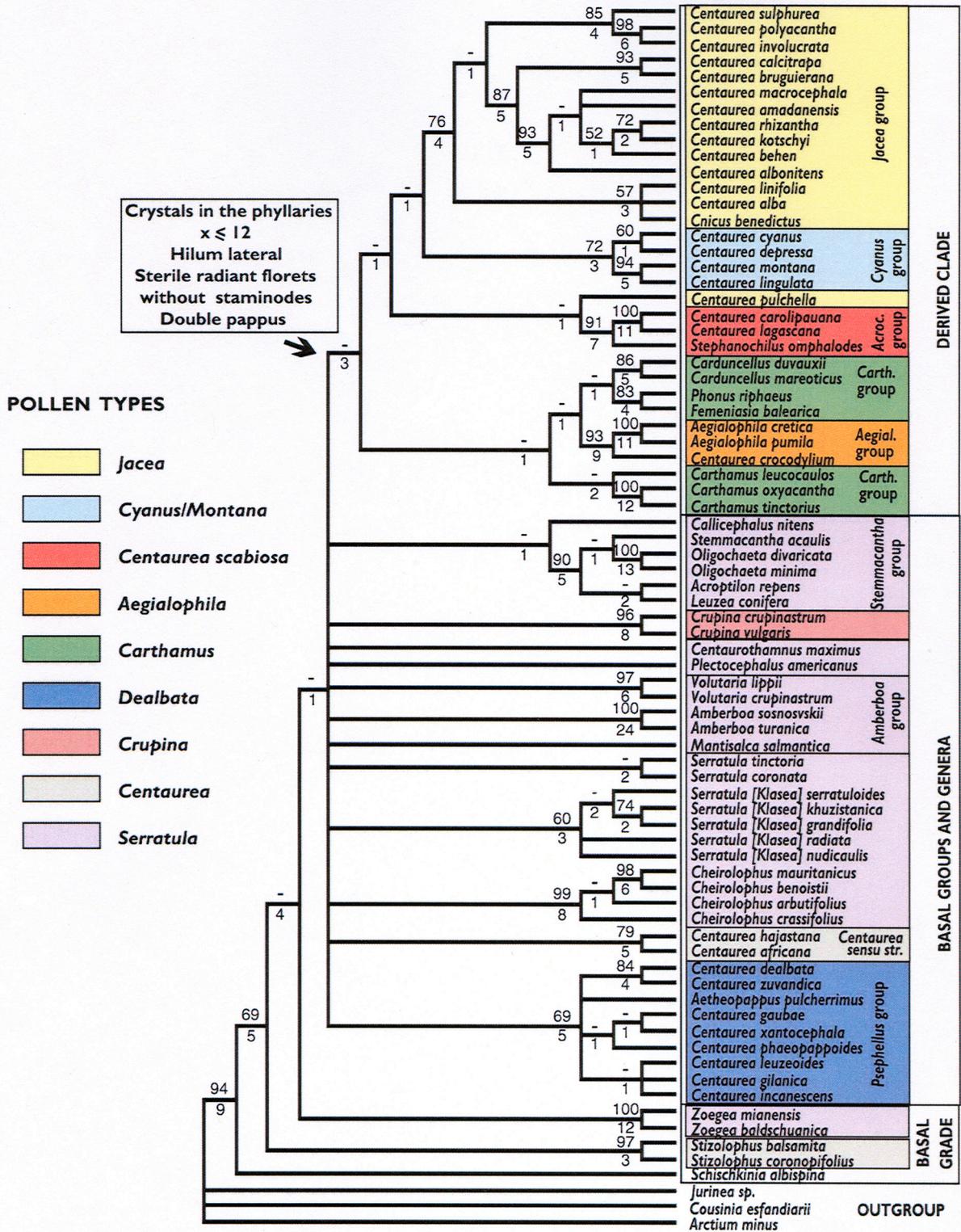


Figure 9. Phylogenetic relationships in the subtribe Centaureinae based on nucleotide sequences of ITS nuclear ribosomal DNA (from Garcia-Jacas *et al.* 2001). Pollen types indicated by color codes are overlaid for comparison to the results of ITS analysis.

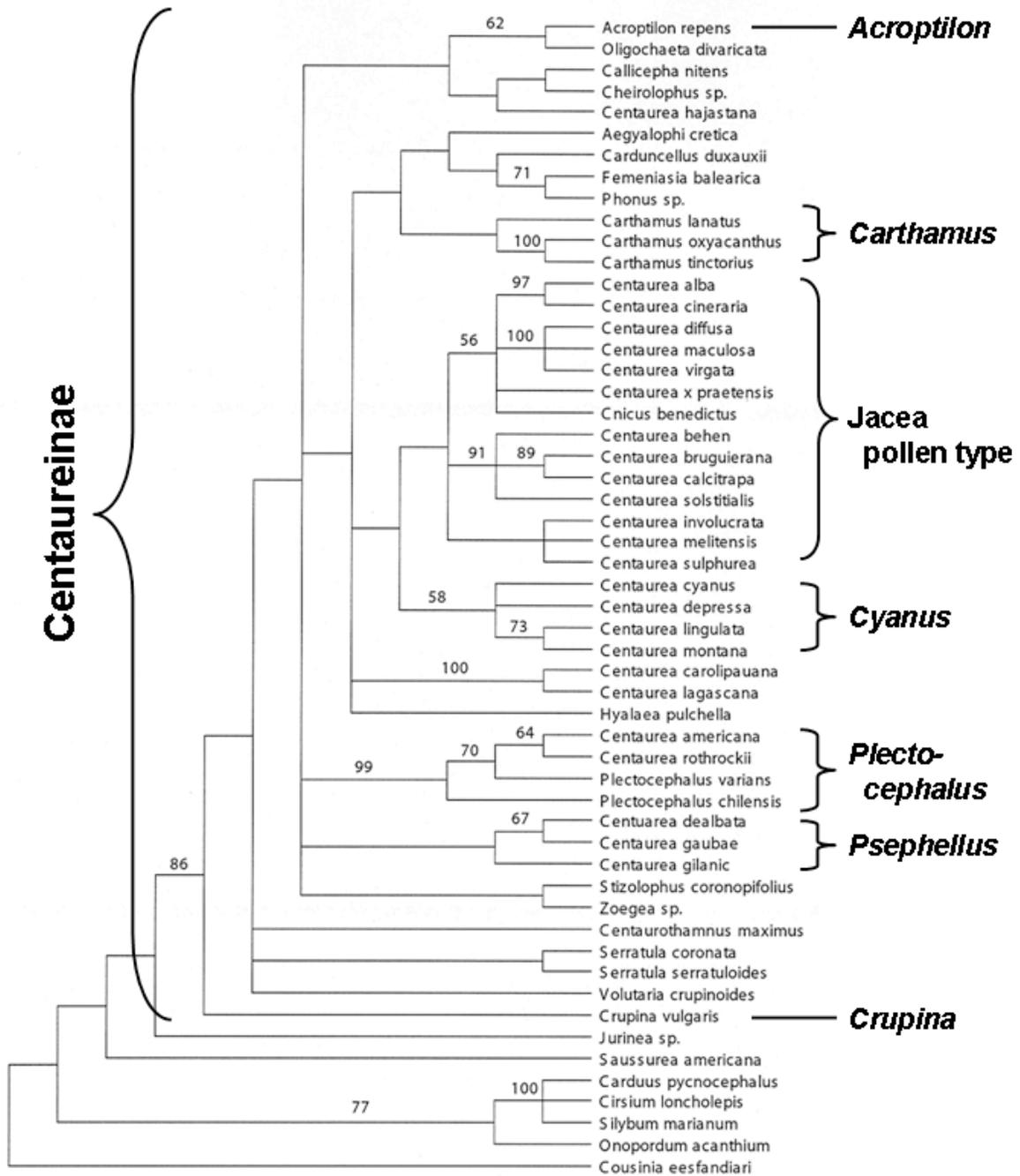


Figure 10. Phylogenetic relationships in the subtribe Centaureinae and some Cardueae relatives based on nucleotide sequences of ITS nuclear ribosomal DNA (J. Gaskin, unpubl. data). Bootstrap values (>50%) written above branches. Currently recognized genera within *Centaurea* (*sensu lato*) and species with Jacea pollen type indicated on the right.

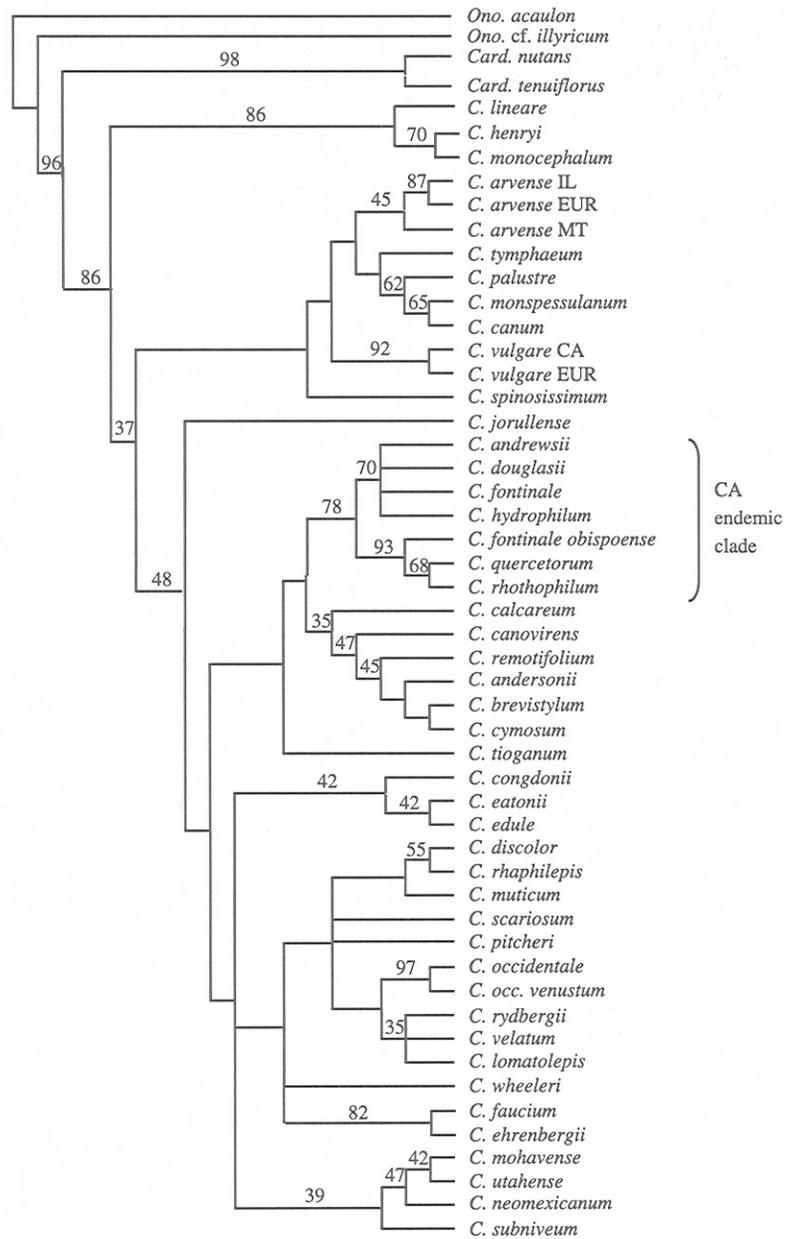


Figure 11. Phylogenetic relationships in the genus *Cirsium* based on maximum likelihood tree of combined ITS and ETS sequence data from nuclear ribosomal DNA (from Kelch and Baldwin 2003). Bootstrap values are written above branches.

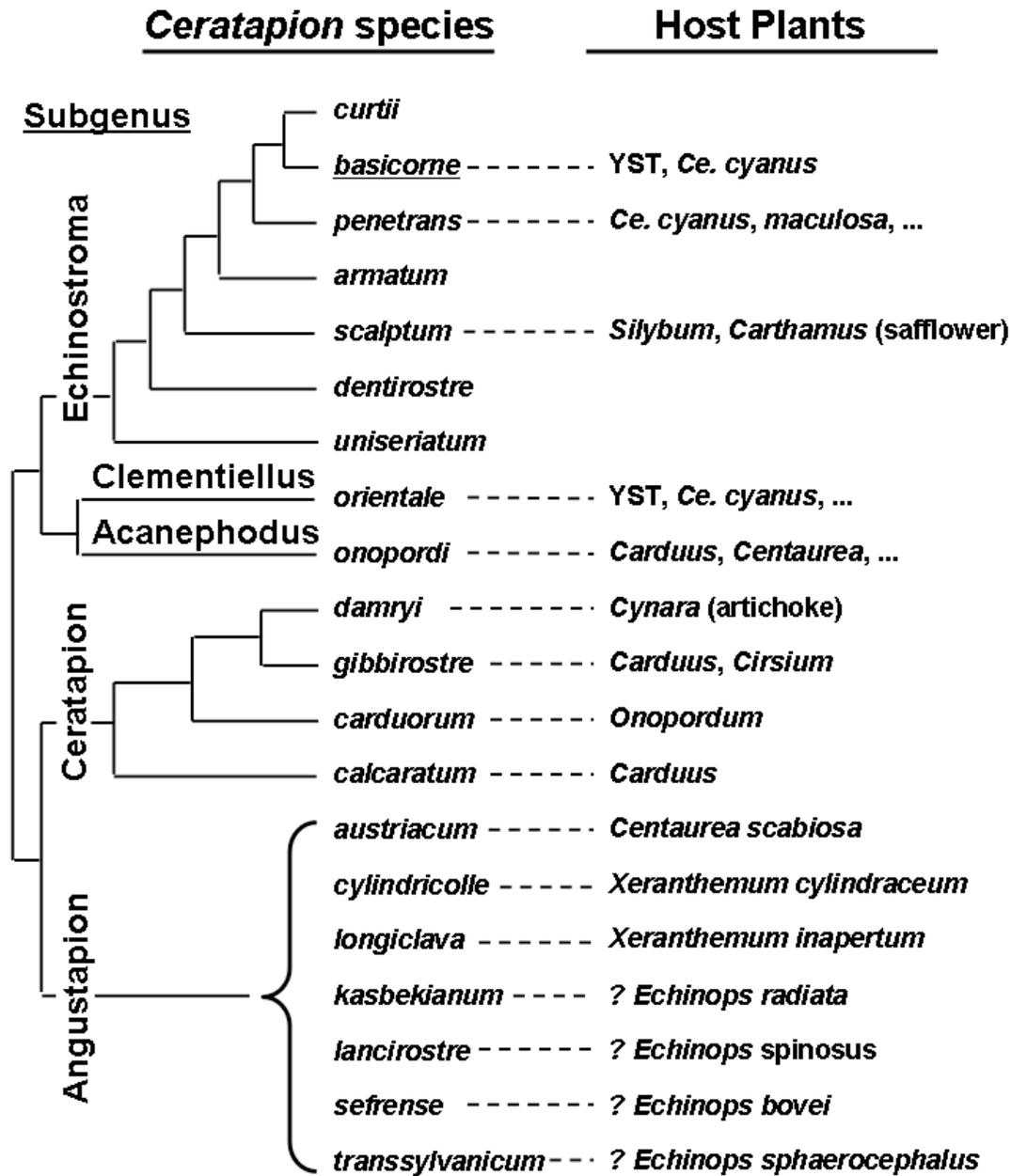


Figure 12. Phylogenetic relationships among species and subgenera in the genus *Ceratapion* after Alonso-Zarazaga (1990a) and Wanat (1994). Larval host records are from both authors. "YST" = yellow starthistle (*Centaurea solstitialis*), "... " = other Cardueae species, "?" = uncertain host plant. Hosts of *C. curtii*, *C. armatum*, *C. dentirostre* and *C. uniseriatum* are unknown.

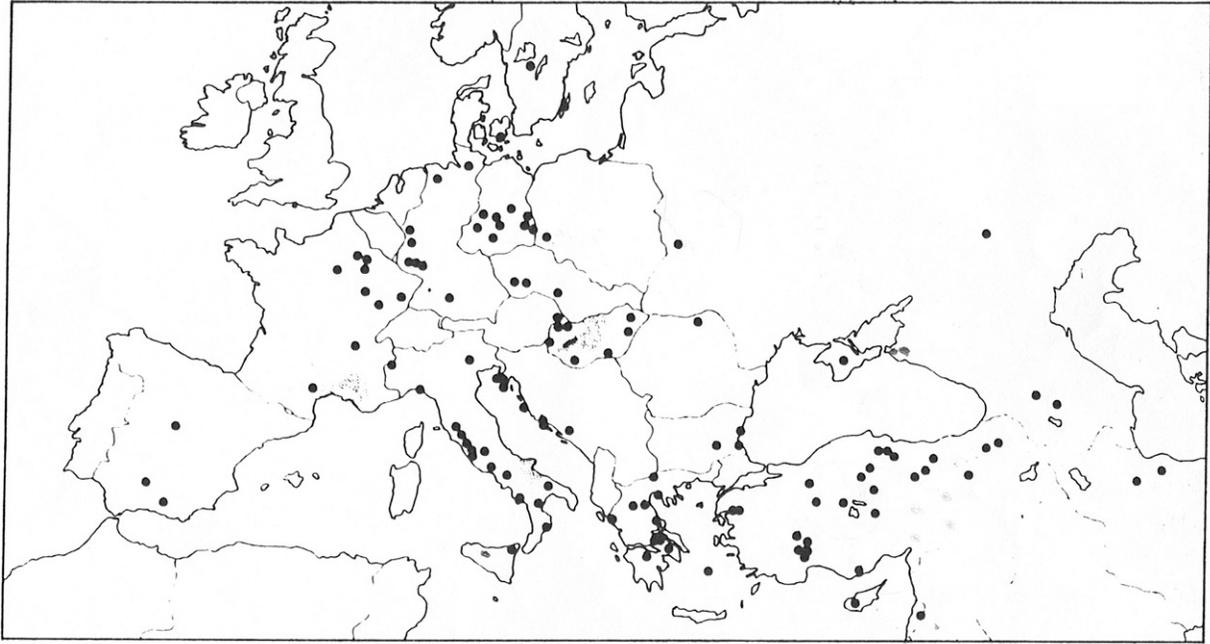


Figure 13. Geographic distribution of *Ceratapion basicorne* in Eurasia (Alonso-Zarazaga 1990a).

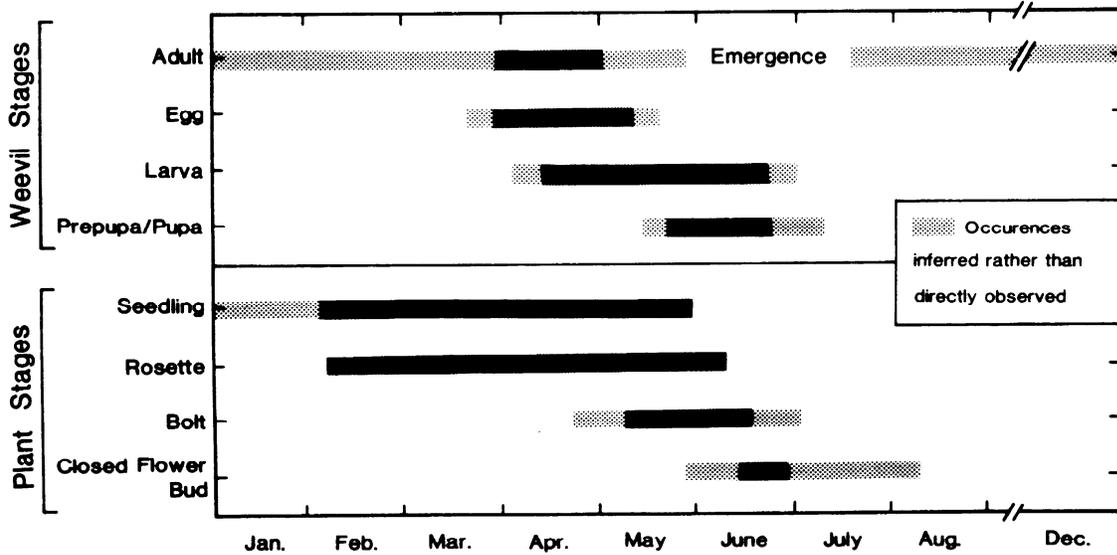
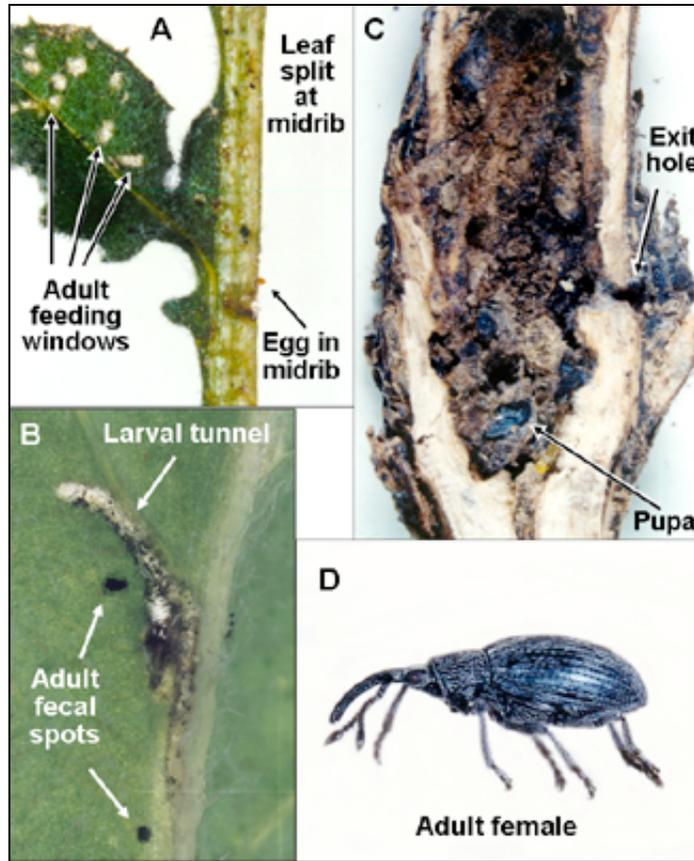


Figure 14. Adult *Ceratapion basicorne* and damage caused by adults feeding on leaves and larvae tunneling in leaves and root crown of yellow starthistle. Seasonal occurrence of *Ceratapion basicorne* in central and southern Italy (from Clement *et al.* 1989).



Figure 15. No-choice oviposition host specificity test. Individual females were held inside a plastic tube attached to the leaf of a test plant for 5 days. Females had to oviposit on yellow starthistle before and after each trial in order for it to be accepted as valid. A small crumpled paper towel provided a hiding place for the insect.



Figure 16. Choice oviposition host specificity tests were conducted in a sleevebox (door open for photograph) inside the quarantine laboratory. Individual females were released in a sleevebox with 4-5 species of test plants (2-4 leaves of the same species in each clump). Tests were run for 5 days. A small crumpled paper towel (center) provided a hiding place for the insect.

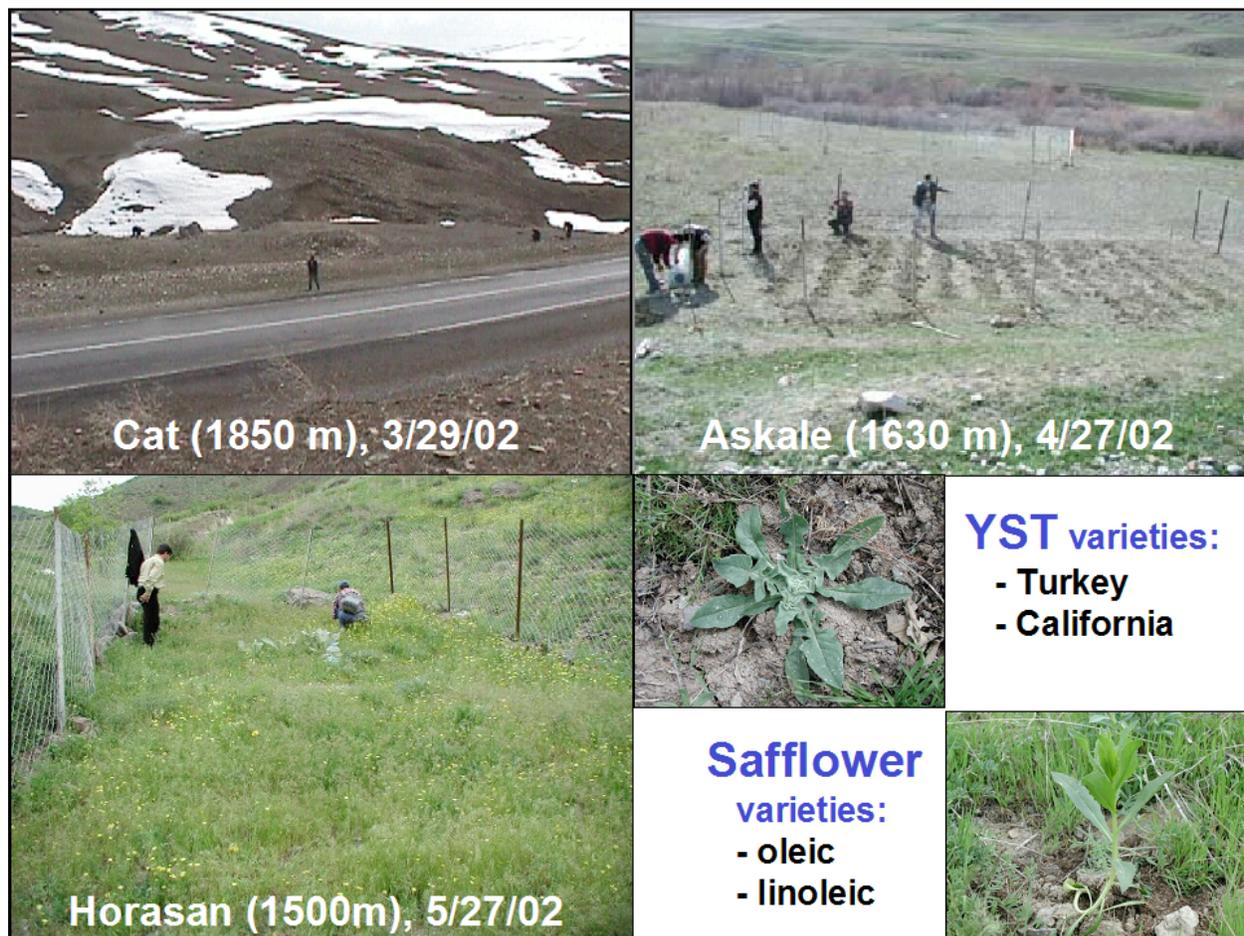


Figure 17. Field studies of host specificity of *Ceratapion basicorne* were conducted at 3 sites in Turkey during 3 years. The weevil was naturally abundant at each site on wild yellow starthistle populations. Test plants of both safflower varieties and yellow starthistle varieties were transplanted into the fields in early spring for exposure to attack by the weevil.



Figure 18. *Ceratopion basicorne* impact study was conducted inside the Albany quarantine laboratory. Yellow starthistle plants at the rosette stage were exposed to ovipositing females (as in no-choice experiments). Then plants were held in the greenhouse until completion of larval development. They were enclosed in screen bags during the last 2 weeks to prevent possible escape of emerging adults. Mean weight of infested plants (bottom left) was 23% lower than of uninfested plants (bottom right).

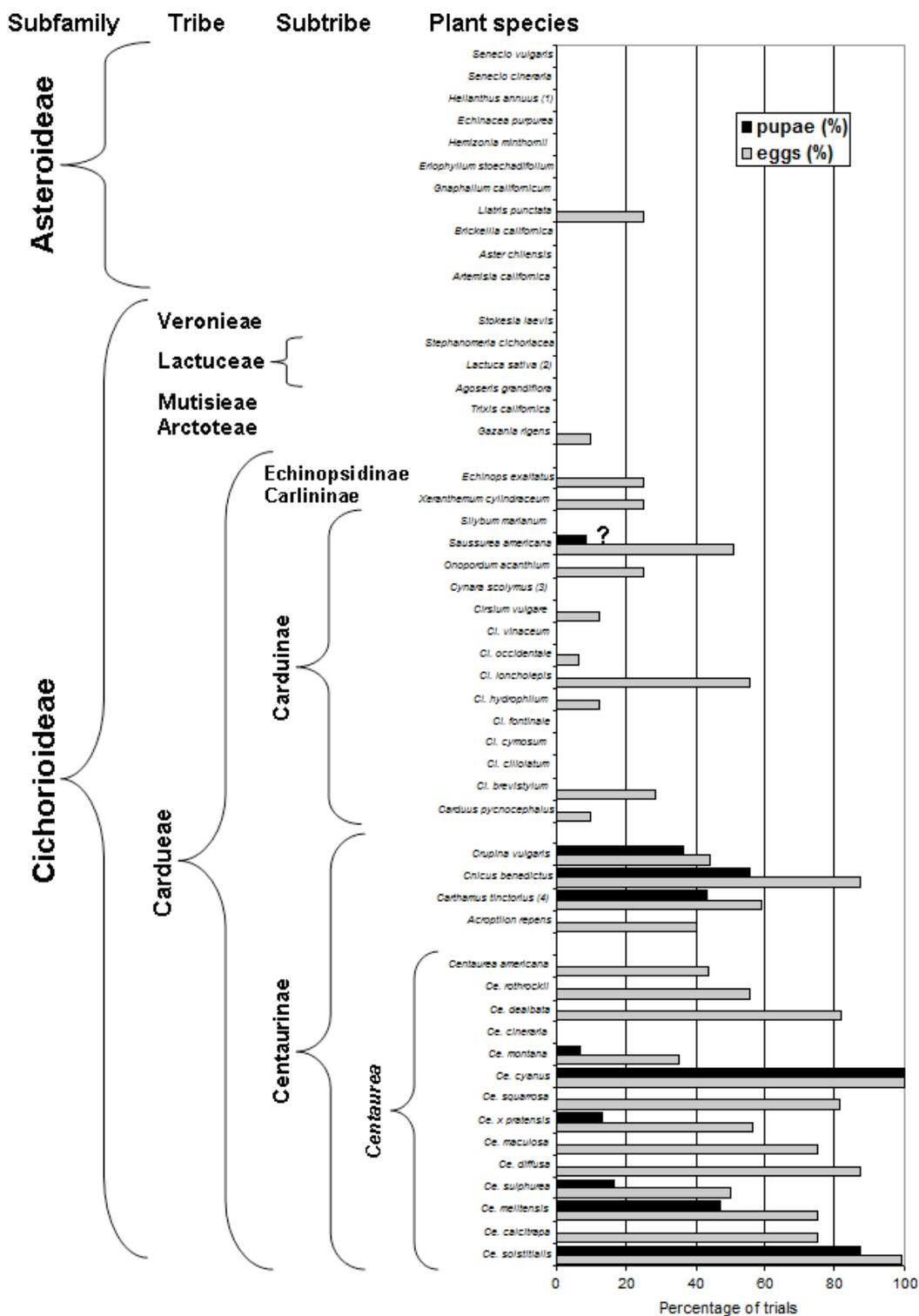


Figure 19. Proportion of trials in which a female oviposited on test plants and in which insects completed development to at least pupal stage in the no-choice host specificity experiment. Individual females were held inside a plastic tube attached to the leaf of a nontarget test plant for 5 days (on yellow starthistle for 2-3 days). (1 - sunflower, 2 - artichoke, 3 - safflower). "?" = damage and exit holes observed in 2 of 23 trials of *Saussurea americana* were probably caused by insects other than *Ceratopion basicorne*.

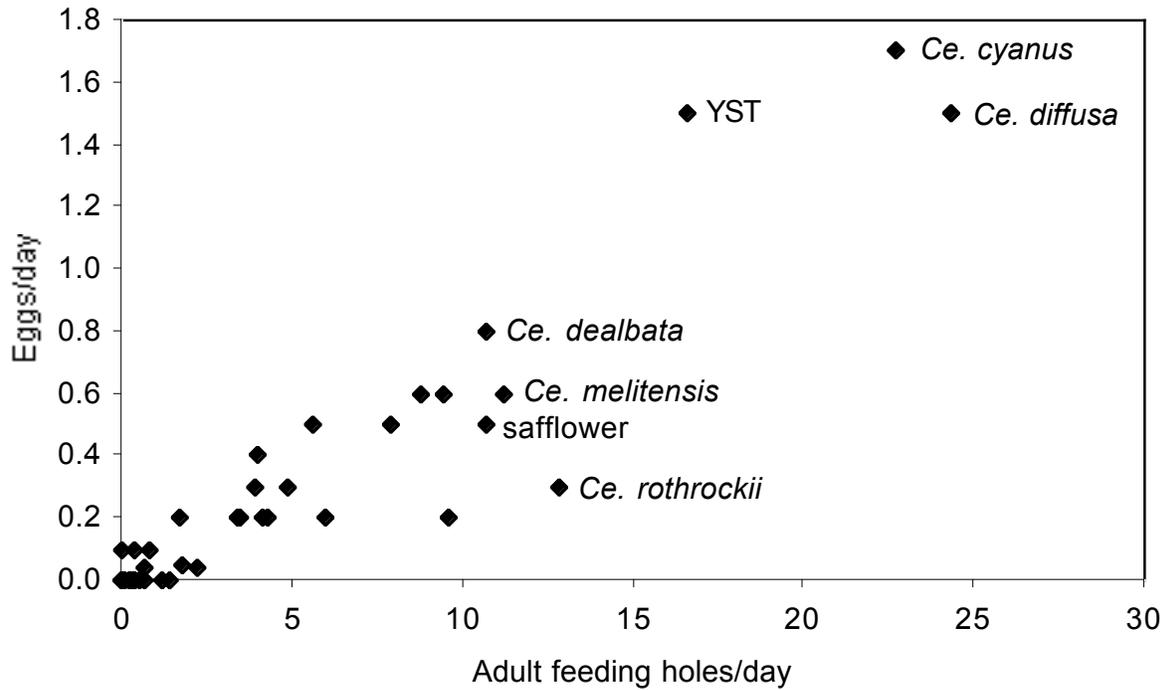


Figure 20. Relationship of adult feeding holes (1-mm^2) to number of eggs on the different test plants in the no-choice oviposition experiment. Each point represents a different test plant species. (YST = *Centaurea solstitialis*, safflower = *Carthamus tinctorius*).

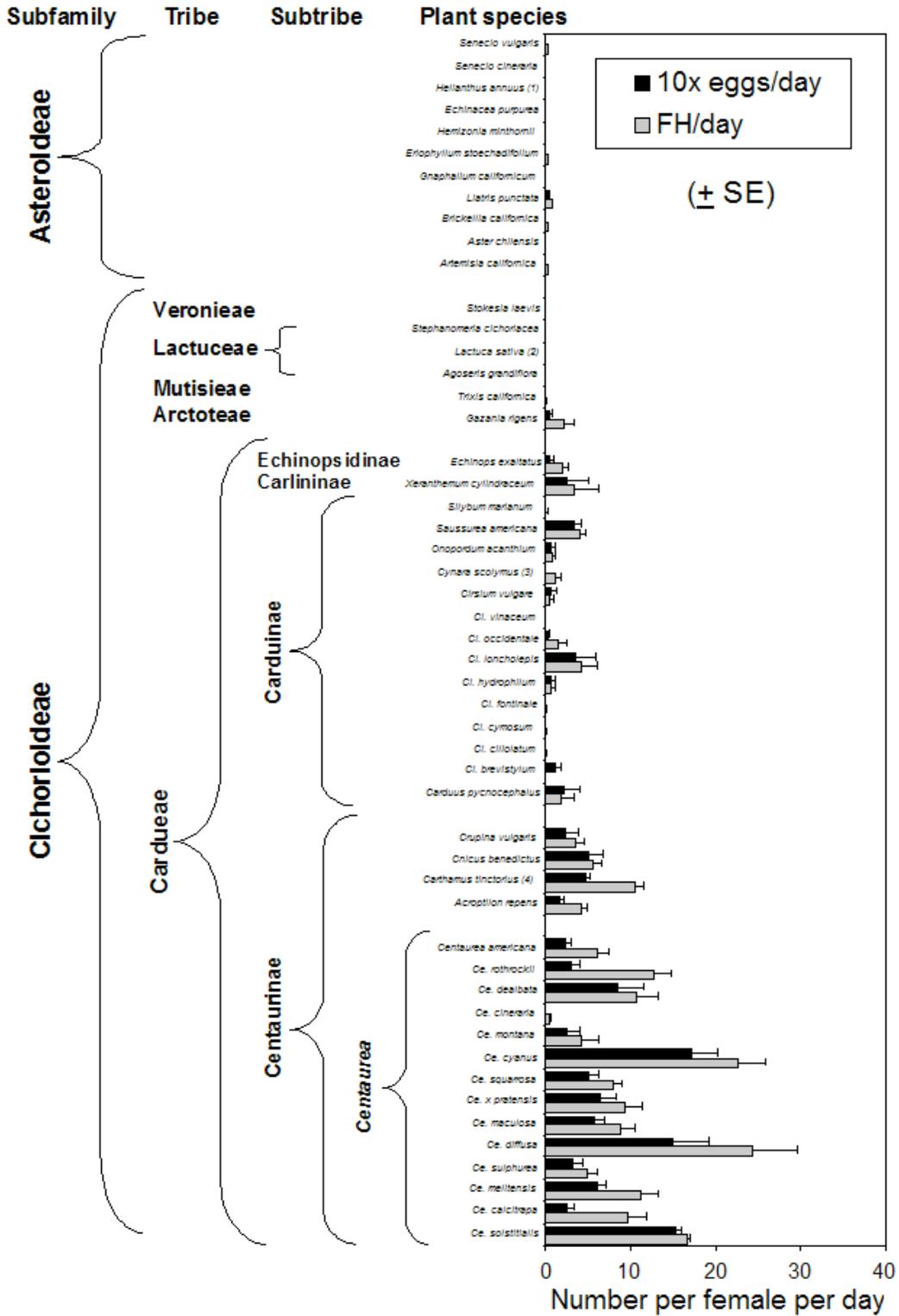


Figure 21. Mean number of eggs (times 10) and mean number of adult feeding holes per day on test plants in the no-choice host specificity experiment. Individual females were held inside a plastic tube attached to the leaf of a nontarget test plant for 5 days (on yellow starthistle for 2-3 days). (error bar = SE, 1 - sunflower, 2 - lettuce, 3 - artichoke, 4 - safflower).

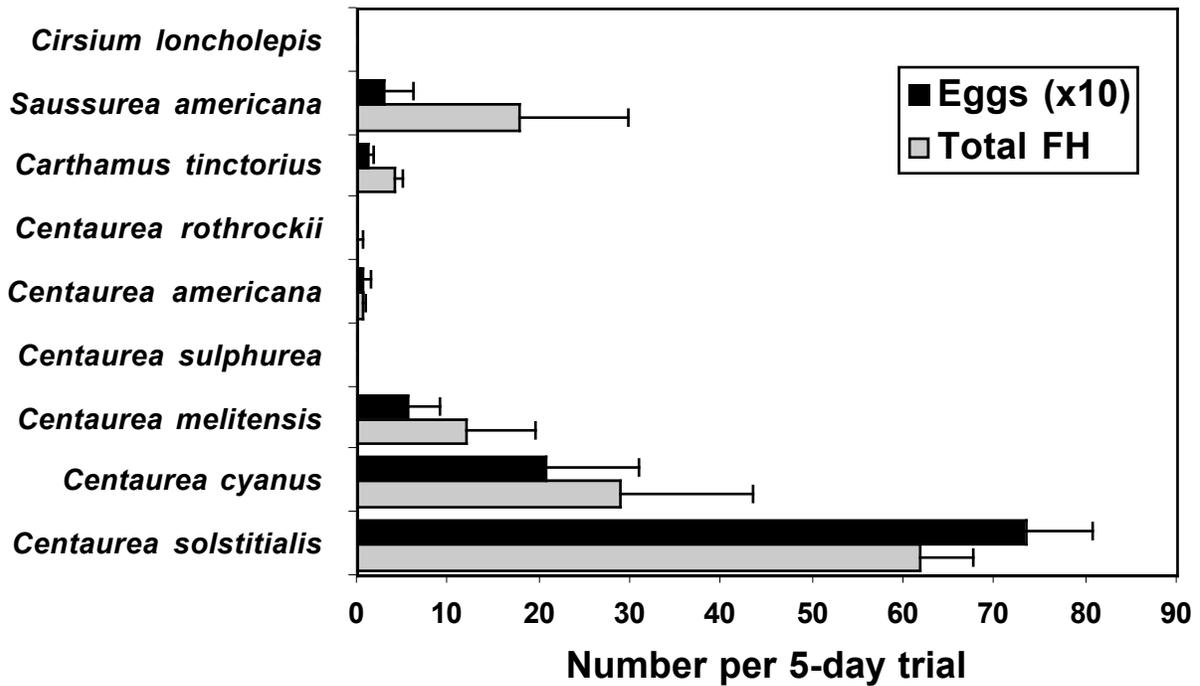


Figure 22. Oviposition and adult feeding by *Ceratapion basicorne* during choice oviposition experiments in sleeveboxes (one female for 5 days exposed to cut leaves of 4-5 plant species at a time, always including yellow starthistle). Number of eggs was multiplied by 10 for visibility on the same scale; FH = number of adult feeding holes, each ca. 1 mm²; error bars = SE. *Car. tinctorius* = safflower, *Ce. cyanus* = bachelor's button, *Ce. solstitialis* = yellow starthistle (target weed).

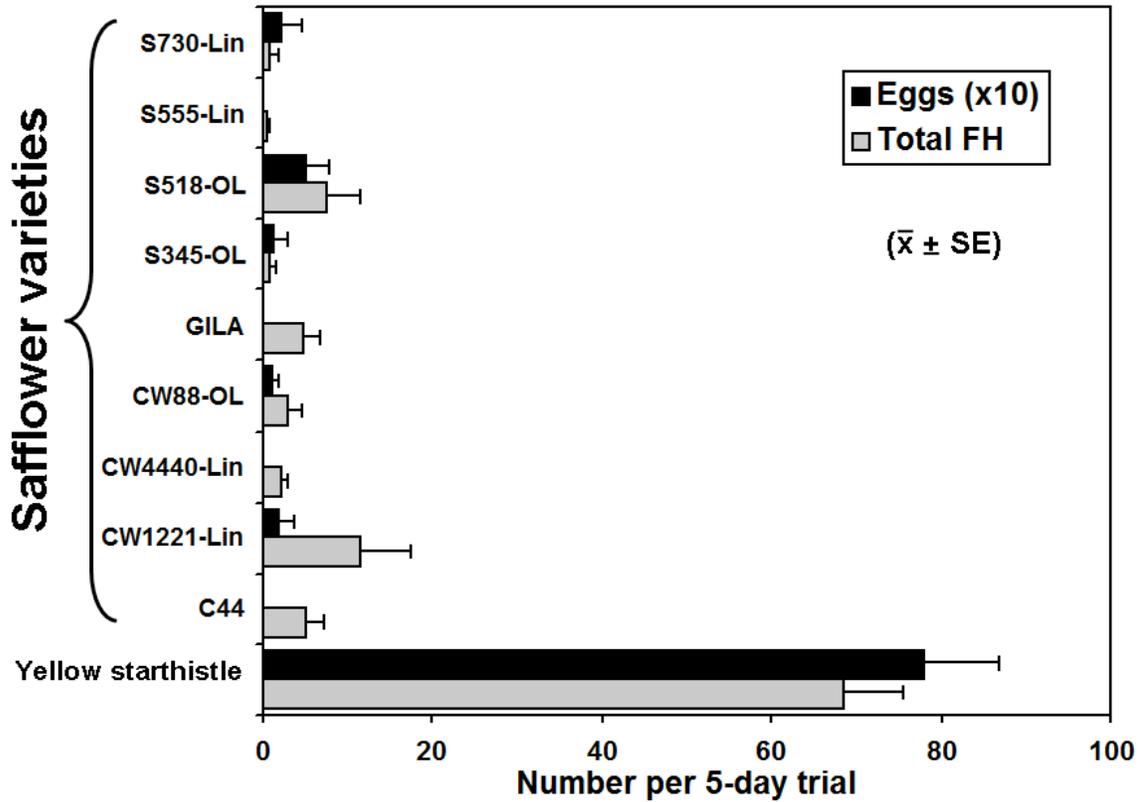


Figure 23. Oviposition and adult feeding by *Ceratopion basicorne* during choice oviposition experiments in sleeveboxes (one female for 5 days exposed to cut leaves of yellow starthistle and 3 safflower varieties at a time). Number of eggs was multiplied by 10 for visibility on the same scale; FH = number of adult feeding holes, each ca. 1 mm². Safflower variety codes: C = Cargill, CW = CalWest, S = Seedtec, Lin = linoleic, OL = oleic.

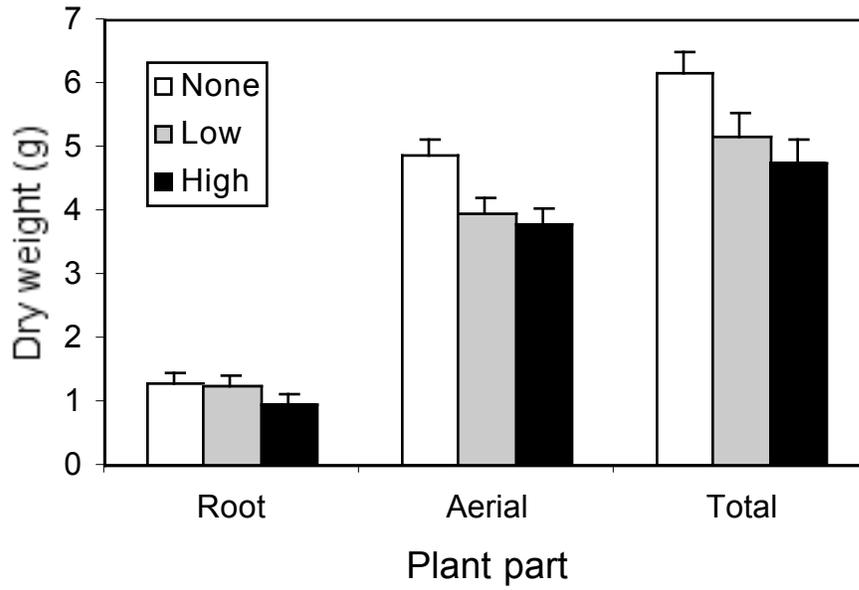


Figure 24. Impact of *Ceratapion basicorne* larval infestation on size of yellow starthistle plants (plants were six weeks old at time of oviposition; Low = 3.1 ± 1.5 (sd) insects per plant, High = 4.2 ± 3.0 insects per plant, error bars = 95% CI).

Appendix 1. Genera and Higher Taxa in the Family Asteraceae occurring in North America.¹

Tribe	Subtribe	Genus	No. species ²		Distribution and notes
			Global	N. Amer.	
Subfamily Cichorioideae					
Arctoteae	Arctotinae			0	Afr, Austr.
Arctoteae	Gorteriinae	Gazania	17	1	ornamental, Africa
Cardueae	Carduinae	Arctium	9	4	Eurasia, weeds
Cardueae	Carduinae	Carduus	90	4	Eurasia, weeds
Cardueae	Carduinae	Cirsium ³	250	100	N Amer, Eurasia, n & e Africa
Cardueae	Carduinae	Cynara ⁴	8	2	crop, weed
Cardueae	Carduinae	Onopordum	60	3	Eurasia, weed
Cardueae	Carduinae	Saussurea ³	300	7	nw N Amer
Cardueae	Carduinae	Silybum	2	1	Eurasia, weed
Cardueae	Carlininae	Xeranthemum	5	1	cultivated
Cardueae	Centaureinae	Acroptilon	1	1	Eurasia, weed
Cardueae	Centaureinae	Carthamus ⁴	17	4	crop, weeds
Cardueae	Centaureinae	Centaurea ^{3,5} (incl. Cnicus)	500	33	Eurasia, weeds
Cardueae	Centaureinae	Crupina	3	1	Eurasia, weed
Cardueae	Echinopsidinae	Echinops	120	7	Eurasia, n&e Africa, ornamental
Lactuceae		Cichorium ⁴	6	2	crop, weed
Lactuceae	Crepidinae	Chondrilla	25	1	alien
Lactuceae	Crepidinae	Crepis ³	200	23	global
Lactuceae	Crepidinae	Taraxacum ³	≥60	9	panarctic
Lactuceae	Dendroseridinae			0	
Lactuceae	Hieraciinae	Hieracium ³	≥90	48	panarctic
Lactuceae	Hypochaeridinae	Hedypnois	2	1	Eurasia, n Africa
Lactuceae	Hypochaeridinae	Hypochaeris	60	4	S. Amer, Europe, Asia
Lactuceae	Hypochaeridinae	Leontodon	50	7	Eurasia, n Africa
Lactuceae	Hypochaeridinae	Picris	40	4	Eurasia, n Africa
Lactuceae	Scorzonerinae	Tragopogon	110	9	Eurasia, n Africa
Lactuceae	Lactucinae	Lactuca ^{3,4}	75	13	crop, Eurasia, Africa, N Amer
Lactuceae	Lactucinae	Nabalus ³	15	13	Asia, N Amer
Lactuceae	Lactucinae	Prenanthes ³	30	15	Eurasia, N Amer
Lactuceae	Malacothricinae	Anisocoma ³	1	1	sw US
Lactuceae	Malacothricinae	Atrichoseris ³	1	1	sw US
Lactuceae	Malacothricinae	Calycoseris ³	2	2	sw US
Lactuceae	Malacothricinae	Glyptopleura ³	2	2	w N Amer

Tribe	Subtribe	Genus	No. species ²		Distribution and notes
			Global	N. Amer.	
Lactuceae	Malacothricinae	Malacothrix ³	16	16	w N Amer
Lactuceae	Malacothricinae	Munzothamnus ³	1	1	California
Lactuceae	Malacothricinae	Pinaropappus ³	10	2	s US, Mexico
Lactuceae	Microseridinae	Agoseris ³	17	8	w N Amer
Lactuceae	Microseridinae	Krigia ³	7	7	c & se US
Lactuceae	Microseridinae	Microseris ³	15	15	w N Amer
Lactuceae	Microseridinae	Nothocalais ³	4	4	c & w N Amer
Lactuceae	Microseridinae	Phalacroseris ³	1	1	w N Amer
Lactuceae	Microseridinae	Pyrrhopappus ³	3	3	N Amer
Lactuceae	Microseridinae	Stebbinsoseris ³	2	2	sw US
Lactuceae	Microseridinae	Uropappus ³	1	1	sw US
Lactuceae	Sonchinae	Sonchus	60	5	Panarctic, Africa, Austr
Lactuceae	Stephanomeriinae	Chaetadelpha ³	1	1	sw US
Lactuceae	Stephanomeriinae	Lygodesmia ³	7	7	w N Amer
Lactuceae	Stephanomeriinae	Prenanthes ³	1	1	sw US
Lactuceae	Stephanomeriinae	Rafinesquia ³	2	2	sw US, Mexico
Lactuceae	Stephanomeriinae	Stephanomeria ³	17	17	w N Amer
Liabeae				0	tropical
Mutiseae	Mutisiinae	Chaptalia ³	60	5	s US, C & S Amer
Mutiseae	Mutisiinae	Gochnatia ³	68	1	s US, C & S Amer
Mutiseae	Mutisiinae	Hecastocleis ³	1	1	US
Mutiseae	Mutisiinae	Leibnitzia ³	6	1	sw US, Mex
Mutiseae	Nassauviinae	Acourtia ³	65	5	s US, Mex, C Amer
Mutiseae	Nassauviinae	Adenocaulon ³	5	1	Asia, N & S Amer
Mutiseae	Nassauviinae	Trixis ³	50	2	sw US, C & S Amer
Vernonieae	Centratherinae			0	tropical
Vernonieae	Elephantopodinae			0	tropical
Vernonieae	Rolandrinae			0	tropical
Vernonieae	Vernoniinae	Stokesia ³	1	1	se US
Vernonieae	Vernoniinae	Vernonia ³	500	30	N & S Amer, Africa, Asia
Subfamily Asteroideae					
Anthemideae	Achilleinae	Achillea ³	115	9	Eurasia, N Africa, a few naturalized in N. Amer.
Anthemideae	Achilleinae	Santolina ³	8	1	Europe, N Africa, intro N Am
Anthemideae	Artemisiinae	Arctanthemum	3	≥1	arctic Eurasia, N Amer
Anthemideae	Artemisiinae	Artemisia ³	390	57	Eurasia, N Amer, Africa, S Amer
Anthemideae	Artemisiinae	Dendranthema ³	37	2	Asia
Anthemideae	Artemisiinae	Hulteniella ³	1	1	arctic Eurasia, N Amer

Tribe	Subtribe	Genus	No. species ²		Distribution and notes
			Global	N. Amer.	
Anthemideae	Artemisiinae	Picrothamnus ³	1	1	w U.S.
Anthemideae	Artemisiinae	Seriphidium ³	130	8	Eurasia, N Amer, N Africa
Anthemideae	Artemisiinae	Sphaeromeria ³	9	9	w U.S., Mex
Anthemideae	Cancriniinae	Tanacetum ³	150	≥5	Eurasia, N. Africa, N Am
Anthemideae	Chrysantheminae	Chrysanthemum	2	7	Europe, Asia, N Africa, some cultivated, weeds
Anthemideae	Leucantheminae	Leucanthemum ³	33	6	Europe, N Africa, cultivated
Anthemideae	Matricariinae	Cotula	55	3	Africa, S Amer, Australia, weeds
Anthemideae	Matricariinae	Matricaria ³	7	4	Eurasia, N Amer, N Africa
Anthemideae	Matricariinae	Soliva	8	2	S Amer, N Amer, Australia
Astereae	Asterinae	Aphanostephus ³	4	4	s US, Mex
Astereae	Asterinae	Aster ³	250	≥2	N hemisphere
Astereae	Asterinae	Baccharis ³	400	23	N Amer, S Amer
Astereae	Asterinae	Boltonia ³	5	5	N Amer
Astereae	Asterinae	Brachyactis ³	6	≥2	N Amer, Asia
Astereae	Asterinae	Callistephus	1	1	Asia, ornamental
Astereae	Asterinae	Chaetopappa ³	10	10	US, Mex
Astereae	Asterinae	Chloracantha ³	1	1	s US, Mex, C Am
Astereae	Asterinae	Conyza ³	60	6	tropical & subtropical, weeds
Astereae	Asterinae	Dichaetophora ³	1	1	s US, Mex
Astereae	Asterinae	Doellingeria ³	7	4	N Amer, Asia
Astereae	Asterinae	Egletes ³	10	2	s US, Mex, S Am
Astereae	Asterinae	Erigeron ³	200	71	N Amer, S Amer
Astereae	Asterinae	Laennecia ³	16	5	sw US, Mex, S Am
Astereae	Asterinae	Monoptilon ³	2	2	sw US, Mex
Astereae	Asterinae	Oreostemma ³	3	3	w US
Astereae	Asterinae	Pentachaeta ³	6	6	sw US, Mex
Astereae	Asterinae	Rigiopappus ³	1	1	sw US, Mex
Astereae	Asterinae	Townsendia ³	25	25	w N Amer, Mex
Astereae	Asterinae	Tracyina ³	1	1	sw US
Astereae	Asterinae	Trimorpha ³	45	3	N Amer, Eurasia
Astereae	Asterinae	Tripolium ³	1	1	N hemisphere
Astereae	Solidagininae	Acamptopappus ³	2	2	sw N Amer
Astereae	Solidagininae	Amphiachyris ³	2	2	US
Astereae	Solidagininae	Amhipappus ³	1	1	sw US
Astereae	Solidagininae	Bigelowia ³	2	2	se US
Astereae	Solidagininae	Chrysoma ³	1	1	se US
Astereae	Solidagininae	Chrysopsis ³	10	10	se US

Tribe	Subtribe	Genus	No. species ²		Distribution and notes
			Global	N. Amer.	
Astereae	Solidagininae	Chrysothamnus ³	15	15	w N Amer
Astereae	Solidagininae	Columbiadorea ³	1	1	nw US
Astereae	Solidagininae	Croptilon ³	3	3	se US
Astereae	Solidagininae	Eastwoodia ³	1	1	sw US
Astereae	Solidagininae	Ericameria ³	27	27	N Amer
Astereae	Solidagininae	Euthamia ³	8	8	N Amer
Astereae	Solidagininae	Grindelia ³	55	28	N Amer, S Amer
Astereae	Solidagininae	Gutierrezia ³	27	8	N Amer, S Amer
Astereae	Solidagininae	Gymnosperma ³	1	1	Texas to C Amer
Astereae	Solidagininae	Hazardia ³	13	7	sw US, Mex
Astereae	Solidagininae	Hesperodoria ³	2	2	sw US
Astereae	Solidagininae	Heterotheca ³	25	25	w US, Mex
Astereae	Solidagininae	Ionactis ³	5	5	N Amer
Astereae	Solidagininae	Isocoma ³	16	10	s US, Mex
Astereae	Solidagininae	Lessingia ³	14	12	sw US, Mex
Astereae	Solidagininae	Machaeranthera ³	36	36	N Amer
Astereae	Solidagininae	Oonopsis ³	3	3	c US
Astereae	Solidagininae	Oreochrysum ³	1	1	w US, Mex
Astereae	Solidagininae	Osbertia ³	2	2	w US, Mex, Can
Astereae	Solidagininae	Petradoria ³	3	3	w US
Astereae	Solidagininae	Pityopsis ³	8	8	se US, Mex
Astereae	Solidagininae	Prionopsis ³	1	1	c & s US
Astereae	Solidagininae	Pyrrocoma ³	10	10	w US, Can
Astereae	Solidagininae	Sericocarpus ³	5	5	N Amer
Astereae	Solidagininae	Solidago ³	150	69	N Amer, Euras, S Am
Astereae	Solidagininae	Stenotus ³	6	6	w US, Can
Astereae	Solidagininae	Thurovia ³	1	1	s US
Astereae	Solidagininae	Tonestus ³	7	7	w U.S., Can
Astereae	Solidagininae	Vancleavea ³	1	1	sw N Amer
Astereae	Solidagininae	Xanthisma ³	1	1	s US
Astereae	Solidagininae	Xanthocephalum ³	5	5	sw US, Mex
Astereae	Solidagininae	Xylorhiza ³	9	9	w US, Mex
Astereae	Solidagininae	Xylothamia ³	9	9	s US, Mex
Calenduleae		Calendula	15	2	Eurasia, N Africa
Eupatorieae	Ageratinae	Ageratum ³	44	6	C & S Amer, some cultivated
Eupatorieae	Ageratinae	Carphochaete ³	7	7	sw U.S., Mexico
Eupatorieae	Ageratinae	Nesomia ³	1	1	Mexico
Eupatorieae	Ageratinae	Piqueriopsis ³	1	1	Mexico

Tribe	Subtribe	Genus	No. species ²		Distribution and notes
			Global	N. Amer.	
Eupatorieae	Ageratinae	<i>Stevia</i> ³	235	8	sw U.S., Mexico, C&S Amer
Eupatorieae	Alomiinae	<i>Ageratella</i>	2	2	Mexico
Eupatorieae	Alomiinae	<i>Alomia</i> ³	5	5	Mexico
Eupatorieae	Alomiinae	<i>Brickellia</i> ³	110	37	w U.S., Mexico, C Amer
Eupatorieae	Alomiinae	<i>Brickelliastrum</i> ³	1	1	s U.S.
Eupatorieae	Alomiinae	<i>Carminatia</i> ³	3	3	Mexico
Eupatorieae	Alomiinae	<i>Flyriella</i> ³	6	6	s U.S., Mexico
Eupatorieae	Alomiinae	<i>Kyrsteniopsis</i> ³	4	4	Mexico
Eupatorieae	Alomiinae	<i>Malperia</i> ³	1	1	sw U.S., Mexico
Eupatorieae	Alomiinae	<i>Pleurocoronis</i> ³	3	3	sw U.S., Mexico
Eupatorieae	Alomiinae	<i>Steviopsis</i> ³	8	8	sw U.S., Mexico
Eupatorieae	Critoniinae	<i>Koanophyllon</i> ³	120	5	s U.S., Mexico, S & C Amer, W Indies
Eupatorieae	Critoniinae	<i>Mexianthus</i> ³	1	1	Mexico
Eupatorieae	Critoniinae	<i>Viereckia</i> ³	1	1	Mexico
Eupatorieae	Eupatoriinae	<i>Eupatorium</i> ³	48	30	e N Amer, W Indies, S & E Asia
Eupatorieae	Fleischmanniinae	<i>Fleischmannia</i> ³	80	4	s U.S. to Argentina
Eupatorieae	Fleischmanniinae	<i>Sartorina</i> ³	1	1	Mexico?
Eupatorieae	Gyptidinae	<i>Conoclinium</i> ³	3	3	e & s U.S., Mexico
Eupatorieae	Gyptidinae	<i>Tamaulipa</i> ³	1	1	s U.S., Mexico
Eupatorieae	Hebecliniinae	<i>Erythradenia</i> ³	1	1	Mexico
Eupatorieae	Hebecliniinae	<i>Matudina</i> ³	1	1	Mexico
Eupatorieae	Hofmeisteriinae	<i>Hofmeisteria</i> ³	12	12	Mexico
Eupatorieae	Liatrinae	<i>Carphephorus</i> ³	7	7	se U.S.
Eupatorieae	Liatrinae	<i>Garberia</i> ³	1	1	se U.S.
Eupatorieae	Liatrinae	<i>Hartwrightia</i> ³	1	1	se U.S.
Eupatorieae	Liatrinae	<i>Liatris</i> ³	43	43	e N Amer, some cultivated
Eupatorieae	Liatrinae	<i>Litrisa</i> ³	1	1	se U.S.
Eupatorieae	Liatrinae	<i>Trilisa</i> ³	2	2	se U.S.
Eupatorieae	Mikaniinae	<i>Mikania</i> ³	430	10	se U.S., pantropical
Eupatorieae	Oxylobinae	<i>Ageratina</i> ³	290	15	N Amer, C Amer, S Amer, W Indies
Eupatorieae	Oxylobinae	<i>Jaliscoa</i> ³	3	3	Mexico
Eupatorieae	Praxelinae	<i>Chromolaena</i> ³	165	9	Americas, some cultivated
Eupatorieae	Trichocoroninae	<i>Sclerolepis</i> ³	1	1	e U.S.
Eupatorieae	Trichocoroninae	<i>Shinnersia</i> ³	1	1	s U.S., Mexico
Eupatorieae	Trichocoroninae	<i>Trichocoronis</i> ³	2	2	sw U.S., Mexico
Gnaphalieae	Cassiniinae	<i>Anaphalis</i> ³	110	1	Asia, N Amer

Tribe	Subtribe	Genus	No. species ²		Distribution and notes
			Global	N. Amer.	
Gnaphalieae	Cassiniinae	Antennaria ³	70	38	N Amer, Eurasia, S Amer
Gnaphalieae	Gnaphaliinae	Ancistrocarphus ³	1	1	sw US
Gnaphalieae	Gnaphaliinae	Bombycilaena	3	?	US, Eurasia
Gnaphalieae	Gnaphaliinae	Filago ³	50	5	N Amer, Euras, n Afr
Gnaphalieae	Gnaphaliinae	Gamochoeta ³	52	6	N Amer, S Amer
Gnaphalieae	Gnaphaliinae	Gnaphalium ³	50	4	global
Gnaphalieae	Gnaphaliinae	Hesperevax ³	3	3	w US
Gnaphalieae	Gnaphaliinae	Pseudognaphalium ³	80	18	S & C Amer, 1 global
Gnaphalieae	Gnaphaliinae	Psilocarphus ³	8	4	Can, US, S Amer
Gnaphalieae	Gnaphaliinae	Stylocline ³	5	5	US, Eurasia
Helenieae		Chaetymenia ³	1	1	Mexico
Helenieae		Coulterella ³	1	1	w Mexico
Helenieae		Dimeresia ³	1	1	w U.S.
Helenieae	Baeriinae	Amblopappus ³	1	1	w U.S., nw Mexico, Peru, Chile
Helenieae	Baeriinae	Baeriopsis ³	1	1	Mexico
Helenieae	Baeriinae	Eatonella ³	1	1	w U.S.
Helenieae	Baeriinae	Eriophyllum ³	11	11	w U.S., nw Mexico, sw Canada
Helenieae	Baeriinae	Lasthenia ³	16	16	Pacific N Amer, Chile
Helenieae	Baeriinae	Lembertia ³	1	1	sw N. Amer
Helenieae	Baeriinae	Monolopia ³	4	4	w U.S.
Helenieae	Baeriinae	Oxypappus ³	2	2	Mexico
Helenieae	Baeriinae	Pseudobahia ³	3	3	w U.S.
Helenieae	Chaenactidinae	Achyropappus	1	1	Mexico
Helenieae	Chaenactidinae	Arnica ³	30	26	N hemisphere
Helenieae	Chaenactidinae	Arnicastrum ³	2	2	Mexico
Helenieae	Chaenactidinae	Bahia ³	13	6	sw U.S., Mexico, Chile
Helenieae	Chaenactidinae	Bartlettia ³	1	1	s U.S., n Mexico
Helenieae	Chaenactidinae	Chaenactis ³	25	25	w U.S., Mexico
Helenieae	Chaenactidinae	Chamaechaenactis ³	1	1	sw U.S.
Helenieae	Chaenactidinae	Espejoa ³	1	1	Mexico, C Amer
Helenieae	Chaenactidinae	Florestina ³	8	1	s U.S., Mexico, Guatemala
Helenieae	Chaenactidinae	Hulsea ³	7	7	w U.S., nw Mexico
Helenieae	Chaenactidinae	Hymenothrix ³	5	5	sw U.S., Mexico
Helenieae	Chaenactidinae	Jamesianthus ³	1	1	s U.S.
Helenieae	Chaenactidinae	Orochaenactis ³	1	1	sw U.S.
Helenieae	Chaenactidinae	Palafoxia ³	12	12	s U.S., Mexico
Helenieae	Chaenactidinae	Peucephyllum ³	1	1	sw U.S., nw Mexico

Tribe	Subtribe	Genus	No. species ²		Distribution and notes
			Global	N. Amer.	
Helenieae	Chaenactidinae	Platyschkuhria ³	1	1	sw U.S.
Helenieae	Chaenactidinae	Psathyrotopsis ³	2	2	Mexico
Helenieae	Chaenactidinae	Schkuhria ³	6	3	N Amer, S Amer
Helenieae	Chaenactidinae	Syntrichopappus ³	2	2	sw U.S.
Helenieae	Chaenactidinae	Venegasia ³	1	1	sw U.S., w Mexico
Helenieae	Chaenactidinae	Whitneya ³	1	1	sw U.S.
Helenieae	Flaveriinae	Clappia ³	1	1	s U.S.
Helenieae	Flaveriinae	Flaveria ³	21	8	s U.S., Mex, C & S Amer, Astralia
Helenieae	Flaveriinae	Haploesthes ³	3	3	s U.S., ne Mexico
Helenieae	Flaveriinae	Jaumea ³	2	1	coasts of w U.S., Mexico, S. Amer
Helenieae	Flaveriinae	Pseudoclappia ³	2	2	s U.S., Mexico
Helenieae	Flaveriinae	Sartwellia ³	4	4	s U.S., ne Mexico
Helenieae	Flaveriinae	Varilla ³	2	2	s U.S., ne Mexico
Helenieae	Gaillardiiinae	Amblyolepis ³	1	1	Mexico
Helenieae	Gaillardiiinae	Baileya ³	4	4	sw U.S., Mexico
Helenieae	Gaillardiiinae	Balduina ³	3	3	se U.S.
Helenieae	Gaillardiiinae	Dugaldia ³	3	3	w U.S. to Guatemala
Helenieae	Gaillardiiinae	Gaillardia ³	28	13	N Amer, S Amer, some cultivated
Helenieae	Gaillardiiinae	Helenium ³	40	19	N Amer, C Amer, S Amer, some cultivated
Helenieae	Gaillardiiinae	Hymenoxys ³	28	18	N Amer, S Amer
Helenieae	Gaillardiiinae	Marshallia ³	7	7	s U.S.
Helenieae	Gaillardiiinae	Plateilema ³	1	1	Mexico
Helenieae	Gaillardiiinae	Plummera ³	2	2	s U.S.
Helenieae	Gaillardiiinae	Psathyrotes ³	3	3	sw U.S.
Helenieae	Gaillardiiinae	Psilostrophe ³	7	7	sw U.S., Mexico
Helenieae	Gaillardiiinae	Trichoptilium ³	1	1	Mexico
Helenieae	Hymenopappinae	Galeana	3	?	Mexico, C Amer
Helenieae	Hymenopappinae	Hymenopappus ³	14	14	N Amer
Helenieae	Hymenopappinae	Loxothysanus ³	3	3	e Mexico
Helenieae	Hymenopappinae	Trichocryne ³	1	1	Mexico
Helenieae	Hymenopappinae	Villanova	10	?	Mexico, C & S Amer
Helenieae	Madiinae	Achyrachaena ³	1	1	w U.S.
Helenieae	Madiinae	Adenothamnus	1	1	nw Mexico
Helenieae	Madiinae	Blepharipappus ³	1	1	w U.S.
Helenieae	Madiinae	Blepharizonia ³	1	1	w U.S.

Tribe	Subtribe	Genus	No. species ²		Distribution and notes
			Global	N. Amer.	
Helenieae	Madiinae	Calycadenia ³	11	11	w U.S.
Helenieae	Madiinae	Hemizonia ³	30	30	U.S., w Mexico
Helenieae	Madiinae	Holocarpha ³	4	4	w U.S.
Helenieae	Madiinae	Holozonia ³	1	1	w U.S.
Helenieae	Madiinae	Lagophylla ³	5	5	w U.S.
Helenieae	Madiinae	Layia ³	15	15	sw U.S., nw Mexico
Helenieae	Madiinae	Madia ³	18	18	w U.S., Chile
Helenieae	Madiinae	Raillardella ³	3	3	w U.S.
Helenieae	Madiinae	Raillardiopsis ³	2	2	sw U.S.
Helenieae	Pectidinae	Adenopappus	1	1	Mexico
Helenieae	Pectidinae	Adenophyllum ³	14	5	sw U.S. to Panama
Helenieae	Pectidinae	Boeberastrum ³	2	2	nw Mexico
Helenieae	Pectidinae	Boeberoides	1	1	Mexico
Helenieae	Pectidinae	Chrysactinia ³	5	5	s U.S., Mexico
Helenieae	Pectidinae	Dysodiopsis ³	1	1	s U.S.
Helenieae	Pectidinae	Dyssodia ³	4	≥1	U.S. to Guatemala
Helenieae	Pectidinae	Gymnolaena ³	3	3	Mexico
Helenieae	Pectidinae	Hydrodyssodia ³	1	1	Mexico
Helenieae	Pectidinae	Hydropectis ³	1	1	Mexico
Helenieae	Pectidinae	Leucactinia ³	1	1	Mexico
Helenieae	Pectidinae	Nicolletia ³	3	3	s U.S., Mexico
Helenieae	Pectidinae	Pectis ³	100	17	N Amer, S Amer
Helenieae	Pectidinae	Porophyllum ³	28	6	N Amer, S Amer
Helenieae	Pectidinae	Strotheria ³	1	1	Mexico
Helenieae	Pectidinae	Tagetes ³	50	8	N Amer, S Amer, some cultivated
Helenieae	Pectidinae	Thymophylla ³	17	17	sw U.S., Mexico
Helenieae	Pectidinae	Urbinella ³	1	1	Mexico
Helenieae	Peritylinae	Eutetras ³	2	2	Mexico
Helenieae	Peritylinae	Pericome ³	4	4	sw N. Amer
Helenieae	Peritylinae	Perityle ³	63	35	w U.S., Mexico, Chile
Heliantheae	(unassigned)	Guardiola ³	10	10	Mexico
Heliantheae	(unassigned)	Stachycephalum	2	?	Mexico, Argentina
Heliantheae	Ambrosiinae	Ambrosia ³	43	28	N Amer, S Amer
Heliantheae	Ambrosiinae	Dicoria ³	4	4	sw U.S., n Mexico
Heliantheae	Ambrosiinae	Euphosyne ³	1	1	Mexico
Heliantheae	Ambrosiinae	Hymenoclea ³	3	3	sw U.S., nw Mexico
Heliantheae	Ambrosiinae	Iva ³	15	15	N Amer

Tribe	Subtribe	Genus	No. species ²		Distribution and notes
			Global	N. Amer.	
Heliantheae	Ambrosiinae	Parthenice ³	1	1	sw U.S., Mexico
Heliantheae	Ambrosiinae	Parthenium ³	16	8	N Amer, C Amer, W Indies
Heliantheae	Ambrosiinae	Xanthium	3	2	global, weeds
Heliantheae	Coreopsidinae	Bidens ³	240	48	global
Heliantheae	Coreopsidinae	Coreocarpus ³	10	10	sw U.S., Mexico
Heliantheae	Coreopsidinae	Coreopsis ³	114	33	N Amer, S Amer, Africa
Heliantheae	Coreopsidinae	Cosmos ³	26	4	Americas, some cultivated
Heliantheae	Coreopsidinae	Dahlia	28	2	Mexico, C & S Amer, some cultivated
Heliantheae	Coreopsidinae	Dicranocarpus ³	1	1	s U.S., Mexico
Heliantheae	Coreopsidinae	Henricksonia ³	1	1	Mexico
Heliantheae	Coreopsidinae	Heterosperma ³	5	1	sw U.S., Mexico, C Amer
Heliantheae	Coreopsidinae	Megalodonta ³	1	1	U.S., Canada
Heliantheae	Coreopsidinae	Thelesperma ³	15	15	w & s N Amer
Heliantheae	Engelmanniinae	Berlandiera ³	4	4	s U.S., Mexico
Heliantheae	Engelmanniinae	Chrysogonum ³	1	1	se U.S.
Heliantheae	Engelmanniinae	Dugesia ³	1	1	Mexico
Heliantheae	Engelmanniinae	Engelmannia ³	1	1	s U.S., n Mexico
Heliantheae	Engelmanniinae	Lindheimera ³	1	1	s U.S., Mexico
Heliantheae	Engelmanniinae	Silphium ³	23	23	U.S.
Heliantheae	Galinsoginae	Alloispermum ³	7	?	N Amer, C Amer, S Amer
Heliantheae	Galinsoginae	Bebbia ³	2	2	sw U.S., nw Mexico
Heliantheae	Galinsoginae	Dyscritothamnus ³	2	2	Mexico
Heliantheae	Galinsoginae	Faxonia ³	1	1	nw Mexico
Heliantheae	Galinsoginae	Galinsoga	13	2	N Amer, C Amer, S Amer
Heliantheae	Galinsoginae	Stenocarpa ³	2	2	Mexico
Heliantheae	Helianthinae	Alvordia ³	4	4	Mexico
Heliantheae	Helianthinae	Helianthus ^{3,4}	50	50	N Amer, some cultivated
Heliantheae	Helianthinae	Heliomeris ³	5	5	w U.S., Mexico
Heliantheae	Helianthinae	Iostephane ³	4	4	Mexico
Heliantheae	Helianthinae	Rhysolepis ³	3	3	Mexico
Heliantheae	Helianthinae	Simsia ³	20	2	s U.S., Mexico, C & S Amer
Heliantheae	Helianthinae	Stuessya ³	3	3	Mexico
Heliantheae	Helianthinae	Tithonia ³	11	3	sw U.S. to Costa Rica
Heliantheae	Helianthinae	Viguiera ³	180	8	N Amer to S Amer
Heliantheae	Melampodiinae	Axiniphyllub ³	5	5	Mexico
Heliantheae	Melampodiinae	Greenmaniella ³	1	1	Mexico
Heliantheae	Melampodiinae	Melampodium ³	37	7	s U.S., Mexico, C & S Amer

Tribe	Subtribe	Genus	No. species ²		Distribution and notes
			Global	N. Amer.	
Heliantheae	Melampodiinae	Polymnia ³	2	2	e N Amer
Heliantheae	Melampodiinae	Smallanthus ³	19	1	e U.S., Mexico, C & S Amer
Heliantheae	Melampodiinae	Tetragonotheca ³	4	4	s U.S., Mexico
Heliantheae	Melampodiinae	Trigonospermum ³	4	4	Mexico
Heliantheae	Melampodiinae	Zandera ³	3	3	Mexico
Heliantheae	Rudbeckiinae	Dracopsis ³	1	1	s U.S.
Heliantheae	Rudbeckiinae	Echinacea ³	9	9	U.S.
Heliantheae	Rudbeckiinae	Ratibida ³	7	7	N Amer
Heliantheae	Rudbeckiinae	Rudbeckia ³	16	16	N Amer
Heliantheae	Verbesininae	Balsamorhiza ³	14	14	w U.S, sw Canada, Mexico
Heliantheae	Verbesininae	Borrichia ³	3	3	se U.S., C Amer, W Indies
Heliantheae	Verbesininae	Calyptocarpus	3	1	s U.S, mexico, C Amer, Cuba, Brazil
Heliantheae	Verbesininae	Chromolepis ³	1	1	Mexico
Heliantheae	Verbesininae	Damxanthodium ³	1	1	Mexico
Heliantheae	Verbesininae	Encelia ³	15	7	sw U.S, Mexico, w S Amer
Heliantheae	Verbesininae	Enceliopsis ³	4	4	sw U.S.
Heliantheae	Verbesininae	Flourensia ³	32	2	s U.S., Mexico, S Amer
Heliantheae	Verbesininae	Geraea ³	2	2	sw U.S., Mexico
Heliantheae	Verbesininae	Helianthella ³	8	8	w U.S., sw Canada, Mexico
Heliantheae	Verbesininae	Hybridella ³	2	2	Mexico
Heliantheae	Verbesininae	Jefea ³	5	≥1	s U.S. to Guatemala
Heliantheae	Verbesininae	Lasianthaea ³	12	1	sw U.S., Mexico, C Amer
Heliantheae	Verbesininae	Melanthera ³	20	5	se U.S., Mexico, C Amer, W Indies, Africa
Heliantheae	Verbesininae	Phoebanthus ³	2	2	se U.S.
Heliantheae	Verbesininae	Tuxtla ³	1	1	Mexico
Heliantheae	Verbesininae	Verbesina ³	300	17	N Amer, C Amer, S Amer
Heliantheae	Verbesininae	Vigethia ³	1	1	Mexico
Heliantheae	Verbesininae	Wyethia ³	14	14	w N Amer
Heliantheae	Verbesininae	Zaluzania ³	10	10	sw U.S., Mexico
Heliantheae	Zinniinae	Heliopsis ³	15	2	N & C Amer, S Amer
Heliantheae	Zinniinae	Philactis	4	?	Mexico, Guatemala
Heliantheae	Zinniinae	Podachaenium	4	?	Mexico, C Amer
Heliantheae	Zinniinae	Sanvitalia ³	7	3	Mexico, C Amer
Heliantheae	Zinniinae	Zinnia	22	?	s U.S., Mexico, C & S Amer
Plucheeae		Pluchea ³	80	10	N & S Amer, tropical
Plucheeae		Pterocaulon ³	18	1	N & S Amer, Austr
Plucheeae		Sachsia ³	4	1	s US, Carib

Tribe	Subtribe	Genus	No. species ²		Distribution and notes
			Global	N. Amer.	
Senecioneae	Blennospermatinae	Blennosperma ³	3	2	CA, Chile
Senecioneae	Blennospermatinae	Crocidium ³	1	1	nw N Amer
Senecioneae	Senecioninae	Erechtites	5	4	N Amer, S Amer
Senecioneae	Senecioninae	Hasteola ³	1	1	e U.S.
Senecioneae	Senecioninae	Packera ³	65	55	N Amer, Siberia
Senecioneae	Senecioninae	Senecio ³	1250	61	global, ornamentals, weeds
Senecioneae	Tussilagininae	Arnoglossum ³	7	7	e & se U.S.
Senecioneae	Tussilagininae	Cacaliopsis ³	1	1	w U.S.
Senecioneae	Tussilagininae	Lepidospartum ³	3	3	sw U.S, Mexico
Senecioneae	Tussilagininae	Ligularia ³	125	8	Eurasia, sometimes cultivated
Senecioneae	Tussilagininae	Luina ³	2	2	w U.S., Canada
Senecioneae	Tussilagininae	Petasites ³	19	5	Eurasia, N Amer
Senecioneae	Tussilagininae	Pippenalia ³	1	1	Mexico
Senecioneae	Tussilagininae	Pittocaulon ³	5	5	Mexico
Senecioneae	Tussilagininae	Psacaliopsis	5	?	Mexico, Guatemala
Senecioneae	Tussilagininae	Psacalium ³	40	1	Mexico, sw U.S, Guatemala
Senecioneae	Tussilagininae	Rainiera ³	1	1	nw US
Senecioneae	Tussilagininae	Roldana ³	55	≥1	Mexico, Guatemala
Senecioneae	Tussilagininae	Rugelia ³	1	1	se U.S.
Senecioneae	Tussilagininae	Telanthophora	14	≥1	Mexico, Guatemala, Honduras
Senecioneae	Tussilagininae	Tephroseris ³	50	4	Eurasia, N. Amer
Senecioneae	Tussilagininae	Tetradymia ³	10	10	w N Amer
Senecioneae	Tussilagininae	Yermo ³	1	1	w N Amer

¹ Based on (Bremer 1994).

² Number of species in world from Bremer (1994), in N. America from Bremer (1994) or PLANTS database (USDA-NRCS 2002).

³ = contains native North American species.

⁴ = contains economically important species.

⁵ = contains target weed species.

Appendix 2. 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. <i>x 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>Liatris 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

¹ *Pluchea odorata* is currently being tested.