Appendix 3A. Brief life histories for biota of concern: Aquatic invertebrates, fishes, and plants

Life history attributes are critical to the analysis and characterization of risk associated with biota transfers, be those biological invasions or shifts in metapopulations that are potentially influenced by interbasin water diversions. While a comprehensive characterization of life history for each biota of concern is precluded by the scope of this investigation and number of biota of concern identified during problem formulation, the life histories that follow are focused on invasive attributes, historical accounts of past invasions, and the current distribution of the species. Existing literature sources in the scientific literature and peer-reviewed public domain provide ample background on each species of interest to the current investigation, with natural resource agencies and the applied research community having disseminated a wealth of data and information for use in these initial investigations of risks associated with biota transfers potentially resulting from interbasin water diversions.

An ability to successfully translocate and accommodate to "new territory" is highly variable across species. Invasiveness depends on attributes related to physiological tolerance, and morphological and behavioral traits that directly or indirectly influence reproductive fitness of the species, thus enabling a species to establish and maintain sustainable populations in a region outside its current distribution. So called, "pioneer species" and invasive species share many of the same, if not identical attributes. Successful expansion of a species' distribution reflects an interrelated multi-step process regardless of whether that distribution expansion results from a "jump dispersal" event common to species invasions mediated directly or indirectly by "hitchhiking," e.g., involving anthropogenic or human cultural means to aid dispersal, or whether that expansion occurs without anthropogenic influences through diffusive mechanisms at the perimeter of existing distributions. Throughout many accounts of species movements and invasions, common attributes have been identified that characterize species as being "invasive" (see, e.g., Elton 1958 for early observations focused on invasive species). For example, Kolar and Lodge (2002) have characterized successful invasions as results of a complex adaptive process or series of events whose outcomes simplify to dependence on (1) transport, (2) establishment, and (3) demographic expansion. While these constituent processes of transport, establishment, and demographic expansion may be decomposed into many interrelated events, such a simplification of the invasion process serves the current investigation and helped focus the data mining activities summarized in the main body of this report and the appendices.

USGS/BRD recognizes the increasing awareness of invasive species and magnitude that species invasions may have on natural resources under the purview of Department of Interior (e.g., Bureau of Land Management, Bureau of Reclamation, National Park Service, and U.S. Fish and Wildlife Service). Invasive species threaten the nation's lands and waterways owing to the adverse effects organisms have on native plants and animals, ecosystems, and the collateral economic effects associated with species invasions (see, e.g., Perrings et al 2000 for overview). The wide range of habitats within the US, and the increased trade and commerce between states and with other countries having similar habitats and ecoregions provides many potential sources and pathways for movements (accidental and intentional) and introductions of biota to areas previously outside a species' distribution.

Invasive species, once established outside their native ecosystems, or repeated, sporadic invasions that may be long-term failures, may play a critical role in the invaded system's structure and function (e.g., sporadic outbreaks of emerging infectious disease may have population-level impacts). Invasive species may be of foreign (exotic species) or domestic origin, with the primary operational attributes shared by these species being their ability to become established in habitats outside their native range. As reflected in the current investigation focused on biota that may be introduced to previously uncharted locations through water diversions, all taxonomic groups – microbes, plants, and animals – include members whose life history attributes favor their being characterized as invasive species. Similar to attributes of invasives that cross-cut many phyla, some ecosystems in the U.S. appear more threatened by invasive species than others. Classically, islands have long been observed as "sensitive" to invasions by a wide assortment of microbes, plants, and animals, but "functional islands" such as habitat fragments, peninsular landforms, and mosaic wetlands exemplify how insular attributes influence the degree of success attained once species have been transported and gain footholds in "new" locations. In the U.S. many habitats are prone to invasion, including low latitude regions of North America characterized by relatively mild climates (e.g., absent extreme seasonal variations in temperature and precipitation), riparian systems across many ecoregions, and western grasslands and range lands. Aquatic habitats throughout the United States are also variously subject to species invasions regardless whether those are lotic or lentic systems. Expanded species distributions in aquatic habitats depend on linkages between sources and surface water habitats such as lakes and ponds, rivers and streams in receiving systems. Despite geographical differences (e.g., differences simply reflecting location or more complicated instances linked to differences between ecoregions characteristic of source and receiving systems), similar pathways may influence physical movement of many potential "invaders" between sources and "target areas" (e.g., open irrigation canals between adjacent low-elevation water sheds). Contrastly, supposedly dissimilar pathways may actually represent relatively subtle differences in mechanisms of species transfer associated with vectors (e.g., dispersal via birds on seasonal migratory routes or overland transport via watercraft used in recreational fishing). Human-influenced pathways and natural pathways lacking human influences may compete as links between source and target areas, as suggested in Annex Figure 1 through Annex Figure 5 in Section 2. More importantly, the simplification of pathways as being "natural" or "human-mediated" may inevitably require recharacterization as a more complex element in the species invasion process when indirect relationships between supposedly independent events are considered (e.g., disturbance may influence the invasibility of habitats).

Biota of concern serving as representative species in USGS/BRD/CERC's interbasin water transfer project completed for U.S. Bureau of Reclamation's Dakota Area Office

An evaluation of risks potentially associated with biota transferred from the Missouri River basin to the Red River basin is daunting, but the risk assessment process, particularly that crafted for the evaluation of ecological risks, is amenable to technical support activities charged with such tasks. Those technical support activities ministering to natural resource managers and their risk management issues, however, must initially reflect a simplification that reflects a phased approach to risk analysis, an approach that varies from application to application, one guided by the stakeholders charged with management and administration responsibility. Hence, in the current investigation, the iterative process of "problem formulation" yielded a menu of biota of concern that were (1) species having a past history of varied discussions regarding their potential roles as "problem species" (see, e.g., http://www.invasivespecies.gov/ for frequently encountered problem species), but more importantly, (2) presented life histories that captured a range of species' attributes that suggested these biota of concern might be representative of species likely to move from one basin to the other as a result of water diversions. Each of these biota of concern – plants, animals, and microorganisms – are acknowledged as being invasive when introduced intentionally or unintentionally by humans and sometimes by natural means. As candidate representative species, these biota have established sustainable populations outside their native ecosystems and have proven to be threats (e.g., economically through displacement of native species) to ecosystems and native species in the US. As developed by Reclamation, stakeholders, and USGS/BRD/CERC through the iterative process of problem formulation, the list of biota of concern included exotic species (i.e., species of foreign origin) and species of domestic origin (i.e., North American), with some of the representative species having become widespread in other regions of the country and having caused significant ecological and economic impacts (e.g., zebra mussel, tamarisk). Through USGS and other resources (private and public such as Center for Disease Control and Prevention) brief life histories have been compiled and edited for Appendix 3A and Appendix 3B. These life histories suggest that existing information and available data for these representative species is documented with sufficient quantity and quality to support an analysis of risks (either qualitative or quantitative) and to help develop adaptive management programs focused on understanding, preventing, monitoring, controlling or eradicating, and predicting the impacts of invasive species in US.

References

Elton, C.S., 1958, The ecology of invasions by plants and animals. The University of Chicago Press, Chicago, Illinois, 181pp.

Kolar, C.S., and D. M. Lodge, 2002, Ecological predictions and risk assessment for alien fishes in North America, Science, 298:1233-1236.

Perrings, C., M. Williamson, and S. Dalmazzone (editors), 2000, The economics of biological invasions, Edward Elgar, Chltenham, UK, 249pp.

Biota of concern: Representative aquatic invertebrates

[1]***Dreissena polymorpha*** **(Pallas 1771)**

Common Name: zebra mussel

Size: Up to 50 mm

Native Distribution. Zebra mussels originated in the Balkans, Poland, and the former Soviet Union. In 1769, Pallas first described populations of this species from the Caspian Sea and Ural River.

Life History. Females generally reproduce in their second year. Fertilization is external and usually occurs in the spring or summer. Optimal temperature for spawning is 14-16 ºC which reflects peak reproduction occurring in early summer at mid-latitude locations. Spawning may last longer in waters that are warm throughout the year, and greater than 40,000 eggs may be laid in a single mating. Multiple matings may occur and up to one million eggs may be fertilized in a spawning season. After the eggs are fertilized, the larvae (veligers) emerge within 3 to 5 days and are free-swimming for up to a month. Optimal temperature for larval development is 20-22 ºC. Dispersal of larvae is normally passive by being carried downstream with the flow. The larvae begin their juvenile stage by settling to the bottom where they crawl about on a foot, searching for suitable substrate for attachment. Once suitable habitat is encountered, the veligers attach themselves to the substrate by means of a multiple-stranded "tether" – the byssus – located outside the body near the foot. Preferred habitats are characterized by hard or rocky substrates and have water velocities less than two meters per second. Although hard substrates are preferred, larvae and juveniles may also attach to vegetation. Zebra mussels are filter feeders, predominately feeding on phytoplankton while filtering nearly a liter of water per day.

Shells of zebra mussels range from dark- to light-colored, having a widely variable striped-pattern to the of their cuticle. Some individuals lack stripes on their shells. Zebra mussels can grow to a maximum length of about 50 mm (5-10 mm in the first year) and live four to five years. They

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[1]Original material accessed September 24, 2002 from USGS/BRD, Center for Aquatic Resource Studies. The Center is part of the Biological Resources Division of the U.S Geological Survey within the U. S. Department of the Interior (last access date, November-December 2004). Updated, expanded, and edited January, 2004.

inhabit fresh water, usually at depths of two to seven meters. Even though zebra mussels are freshwater animals, they have recently been found living in brackish water with salinity levels of one to two parts per thousand. Calcium is an important nutrient for survival with an optimal range of 25 -125 parts per million. Other environmental factors such as water temperature, pH, and dissolved oxygen also play a critical role in survival. Water temperatures of 17-25 ºC are optimal for survival across all life stages (veliger, juvenile, and adult). Temperature exceeding 30-31 ºC is usually lethal, and waters characterized by pH less than 7.4 or greater than 8.5 are commonly not sufficient to support growth and reproduction. Well-oxygenated waters having 8-10 parts per million dissolved oxygen are preferred. Under cool, humid conditions, zebra mussels can stay alive for several days out of water.

Nonindigenous Occurrences. Zebra mussels apparently originated in eastern Europe (e.g., Balkans, Poland, and Ukraine) but had spread to most all major drainages of Europe between the 1600's and 1700's because of widespread construction of canal systems. Great Britain was invaded in 1824 and currently has well-established populations of zebra mussel. Throughout the 19^{th} and 20^{th} centuries, zebra mussels have expanded throughout western Europe and countries of Scandinavia.

Figure 1 and Figure 2 summarize the geographic distribution of zebra mussels in North America. In 1988, zebra mussels were first recorded in an account of an established population in the Canadian waters of Lake St. Clair, a small water body connecting Lake Huron and Lake Erie. By 1990, zebra mussels occurred throughout all the Great Lakes, and in 1991, zebra mussels escaped the Great Lakes basin and found their way into the Illinois and Hudson rivers. The Illinois River was the key to their introduction into the Mississippi River drainage which covers over 1.2 million square miles. By 1992, the following rivers had established populations of zebra mussels: Arkansas, Cumberland, Hudson, Illinois, Mississippi, Ohio, and Tennessee. By 1994, the following states had reported records of zebra mussels within their borders or in water bodies adjacent to their borders: Alabama, Arkansas, Illinois, Indiana, Iowa, Kentucky, Louisiana, Michigan, Minnesota, Mississippi, Missouri, New York, Ohio, Oklahoma, Pennsylvania, Tennessee, Vermont, West Virginia, and Wisconsin. More recently, Connecticut has been added to the list of states where zebra mussels have been found. In 2002, zebra mussels were first recorded in a small isolated quarry in Virginia, and in 2003 Kansas and South Dakota recorded their first sightings of zebra mussels.

Method of Introduction. It is highly likely that the presence of zebra mussels in the Great Lakes was a result of a ballast water introduction, and its dispersal throughout the Great Lakes and major river systems occurred relatively rapidly due to its ability to attach to boats navigating these lakes and rivers. Its rapid range expansion into interconnected waterways was probably due to barge traffic where it is theorized that attached mussels were scraped or fell off during routine navigation. Overland dispersal is also a strong possibility for aiding zebra mussel range expansion (see, e.g., Johnson et al 2001), and many small lakes in the Great Lakes basin have likely been accessed by mussels "hitch-hiking" as veligers and juveniles attached to watercraft transported from infested waters to uninfested waters where populations of zebra mussels have subsequently become established. Haphazard inspections throughout North America have found zebra mussels attached to hulls or in motor compartments of watercraft, including a documented observation outside Regina, Manitoba.

Ecological and economic impacts. Zebra mussels are notorious for their biofouling capabilities by colonizing water supply pipes of hydroelectric and nuclear power plants, public water supply plants, and industrial facilities (see, e.g., D'Itri 1997, Nalepa and Schloesser 1993). As veligers and juveniles, the species colonizes pipes and other hard substrates, then as adults, the established colonies reduce intake and restrict flow in water distribution and treatment systems, heat exchangers, condensers, fire fighting equipment, and air conditioning and cooling systems. For example, populations densities for zebra mussel have been recorded as high as 700,000 m^2 at power plants (e.g., in Michigan) and the diameters of pipes have been reduced by two-thirds at water treatment facilities. Although there is little information on zebra mussels affecting irrigation systems, farms and golf courses could be likely candidates for infestations, if their source waters are infested.

Navigational and recreational boating can be affected by increased drag due to attached mussels. Small mussels can get into engine cooling systems causing overheating and damage. Navigational buoys have been sunk under the weight of attached zebra mussels, and fishing gear can be fouled if left in the water for long periods. Deterioration of dock pilings has increased when they are encrusted with zebra mussels, and continued attachment of zebra mussel can cause corrosion of steel and concrete affecting its structural integrity.

Most of the biological impacts of zebra mussels in North America are poorly characterized, especially those indirect effects at higher levels of biological organization and those direct effects that stem from interactions with multiple-species in community settings. However, information from Europe tells us that zebra mussels have the potential to severely impact unionids (native mussels) by interfering with their feeding, growth, locomotion, respiration, and reproduction. Researchers are observing some of these effects as they study interactions between zebra mussels and native unionids in the Great Lakes. In one study they determined that where zebra mussel densities were highest in Lake St. Clair and in the western basin of Lake Erie, the number of native unionids had dramatically declined (Schloesser and Nalepa 1994). This difference was seen after only two years of zebra mussel colonization. Other studies have shown an inverse correlation between zebra mussel biomass and unionid density (Nalepa 1994). Scientists in the Great Lakes region have been using models that may predict the degree of unionid mortality based on zebra mussel densities (Ricciardi 1995). Unfortunately, research shows zebra mussels prefer to attach to live unionids rather than to dead ones or to rocks (Schloesser and Kovalak 1991). Some unionids have been found with more than 10,000 zebra mussels attached to them. This represents a tripling or even a quadrupling of the unionid's own weight (Hebert et al. 1991). Native unionids may not survive if zebra mussels continue to colonize Lake St. Clair (Hunter and Bailey 1992). The St. Croix River, a federally designated wild and scenic river in the upper Mississippi River basin, is being heavily guarded by the National Park Service because it contains the only known viable population of the winged mapleleaf clam (*Quadrula frugosa*). Zebra mussels could wipe out these clams if they become established in the river. Placing the native unionids in temporary refugia or transplanting them in waters absent of zebra mussels is being used as an alternative to try and save them. Another exotic invader, the quagga mussel (*Dreissena bugensis*), probably arrived at the same time as the zebra mussel. Although the quagga mussel closely resembles its cousin, it is not expected to have as great an impact on unionids because it does not show a preference for unionids as substrates (Conn and Conn 1993).

According to early studies, zebra mussels are having a minimal effect on fish populations in the Great Lakes. It may be too soon to determine some of the effects which may take more time to develop. However, there has been a striking difference in water clarity improving dramatically in Lake Erie, sometimes four to six times what is was before the arrival of zebra mussels. With this increase in water clarity, more light is able to penetrate deeper allowing for an increase in aquatic plants (Skubinna et al. 1995). Some of these macrophyte beds have not been seen for many decades due to changing conditions of the lake mostly due to pollution. The macrophyte beds that have returned are providing cover and acting as nurseries for some species of fish.

Methods of Control. There are many methods that have been investigated to help control zebra mussels, and these listed below in no particular order. Some methods will work better than others in a particular situation.

- Chemical Molluscicides: Oxidizing (chlorine, chlorine dioxide) and non-oxidizing chemical treatments
- Manual Removal (pigging, high pressure wash)
- Dewatering/Desiccation (freezing, heated air)
- Thermal (steam injection, hot water > 32 oC)
- Acoustical Vibration
- Electrical Current
- Filters, Screens
- Coatings: Toxic (copper, zinc) and Non-toxic (silicone-based)
- Toxic Constructed Piping (copper, brass, galvanized metals)
- CO_2 Injection
- Ultraviolet Light
- Anoxia/Hypoxia
- Flushing
- Biological (predators, parasites, diseases)

References

Conn, D. B., and D. A. Conn. 1993. Parasitism, predation, and other associations between Dreissenid mussels and other native animals in the St. Lawrence River. Proceedings: Third International Zebra Mussels Conference, 1993. Electric Power Research Institute pp. 2-25 - 2-34.

D'Itri, F.M. (editor), 1997, Zebra mussels and aquatic nuisance species, Lewis Publishers, Boca Raton, Florida, 638pp.

Hebert, P. D. N., C. C. Wilson, M. H. Murdoch, and R. Lazar. 1991. Demography and ecological impacts of the invading mollusc *Dreissena polymorpha*. Can. J. Zool. 69:405-409.

Hunter, R. D., and J. F. Bailey. 1992. *Dreissena polymorpha*(zebra mussel): colonization of soft substrata and some effects on unionid bivalves. The Nautilus 106(2):60-67.

Johnson, L.E., A. Ricciardi, and J.T. Carlton, 2001, Overland dispersal of aquatic invasive species: A risk assessment of transient recreational boating, Ecological Applications 11:1789-1799.

Nalepa, T. F. 1994. Decline of native unionid bivalves in Lake St. Clair after infestation by the zebra mussel, *Dreissena polymorpha*. Can. J. Fish. Aquat. Sci. 51:2227-2233.

Nalepa, T.F., and D.W. Schloesser (editors), 1993, Zebra mussels: Biology, impacts, and control, Lewis Publishers, Boca Raton, Florida, 810pp.

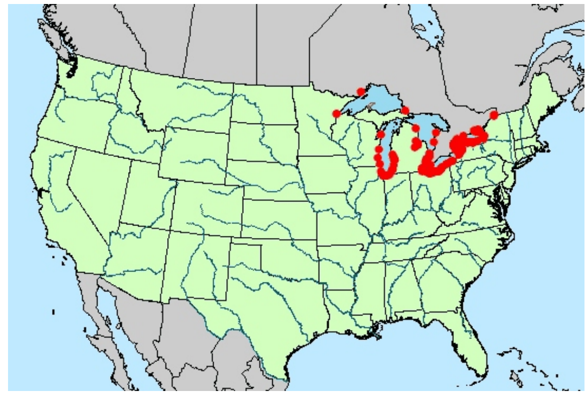
Ricciardi, A., F. G. Whoriskey, and J. B. Rasmussen. 1995. Predicting the intensity and impact of *Dreissena*infestation native unionid bivalves from *Dreissena* density. Can. J. Fish. Aquat. Sci. 52:1449-1461.

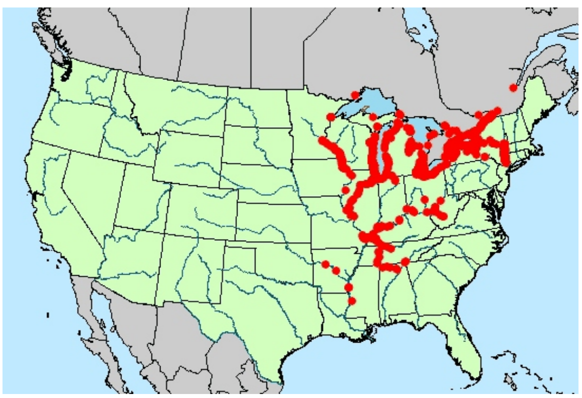
Schloesser, D. W., and T. F. Nalepa. 1994. Dramatic decline of native unionid bivalves in offshore waters of western Lake Erie after infestation by the zebra mussel, *Dreissena polymorpha*. Can. J. Fish. Aquat. Sci. 51:2234-2242.

Skubinna, J.P., T.G. Coon, and T.R. Batterson. 1995. Increased abundance and depth of submersed macrophytes in response to decreased turbidity in Saginaw Bay, Michigan. Journal of Great Lakes Research 21(4): 476-488.

Figure 1. Distribution maps illustrating the progressive invasion of zebra mussel.


1988


1990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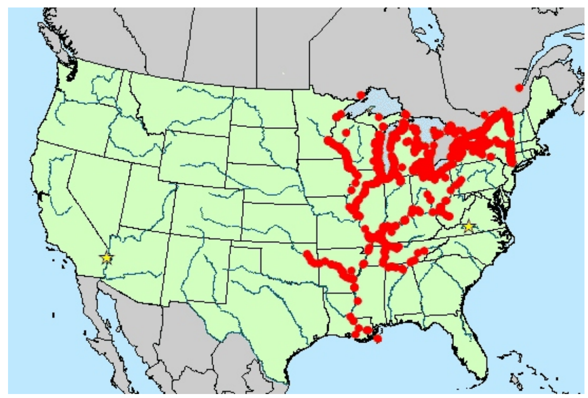
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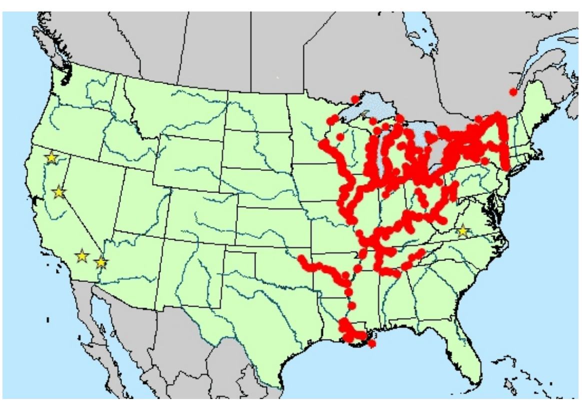
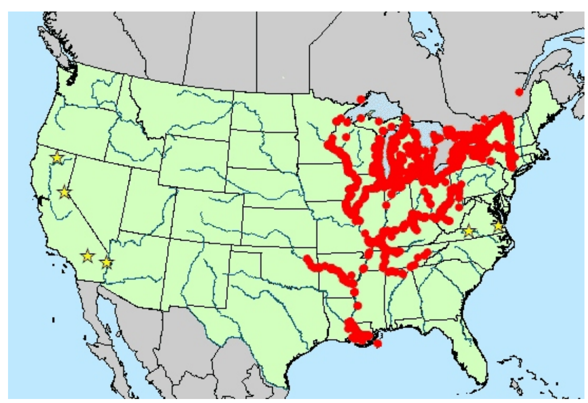
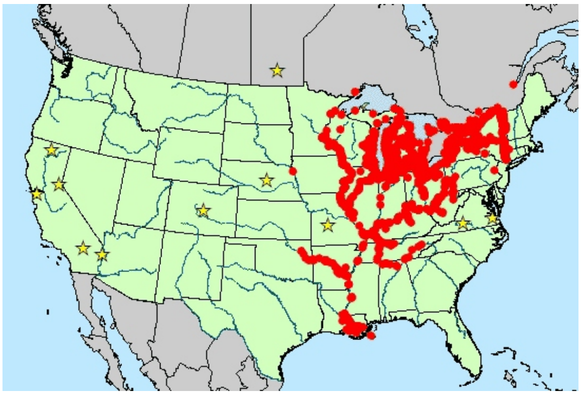

1996


1998



2000

2002

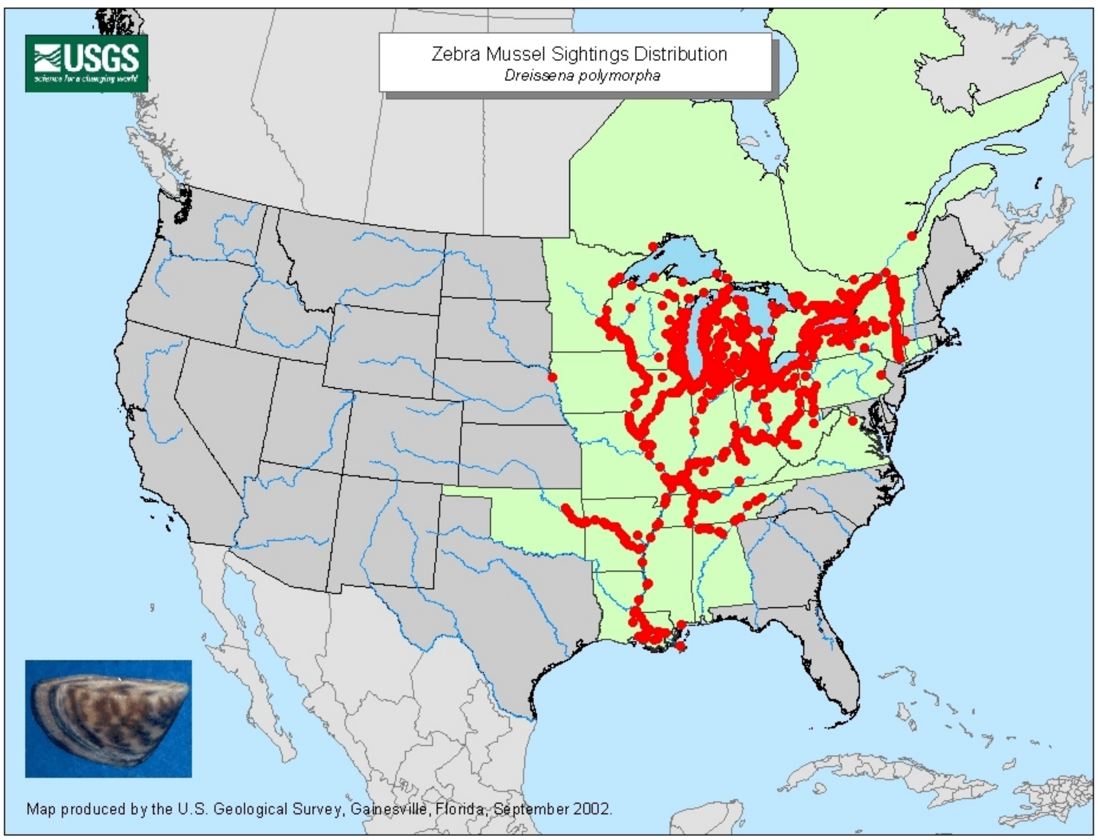
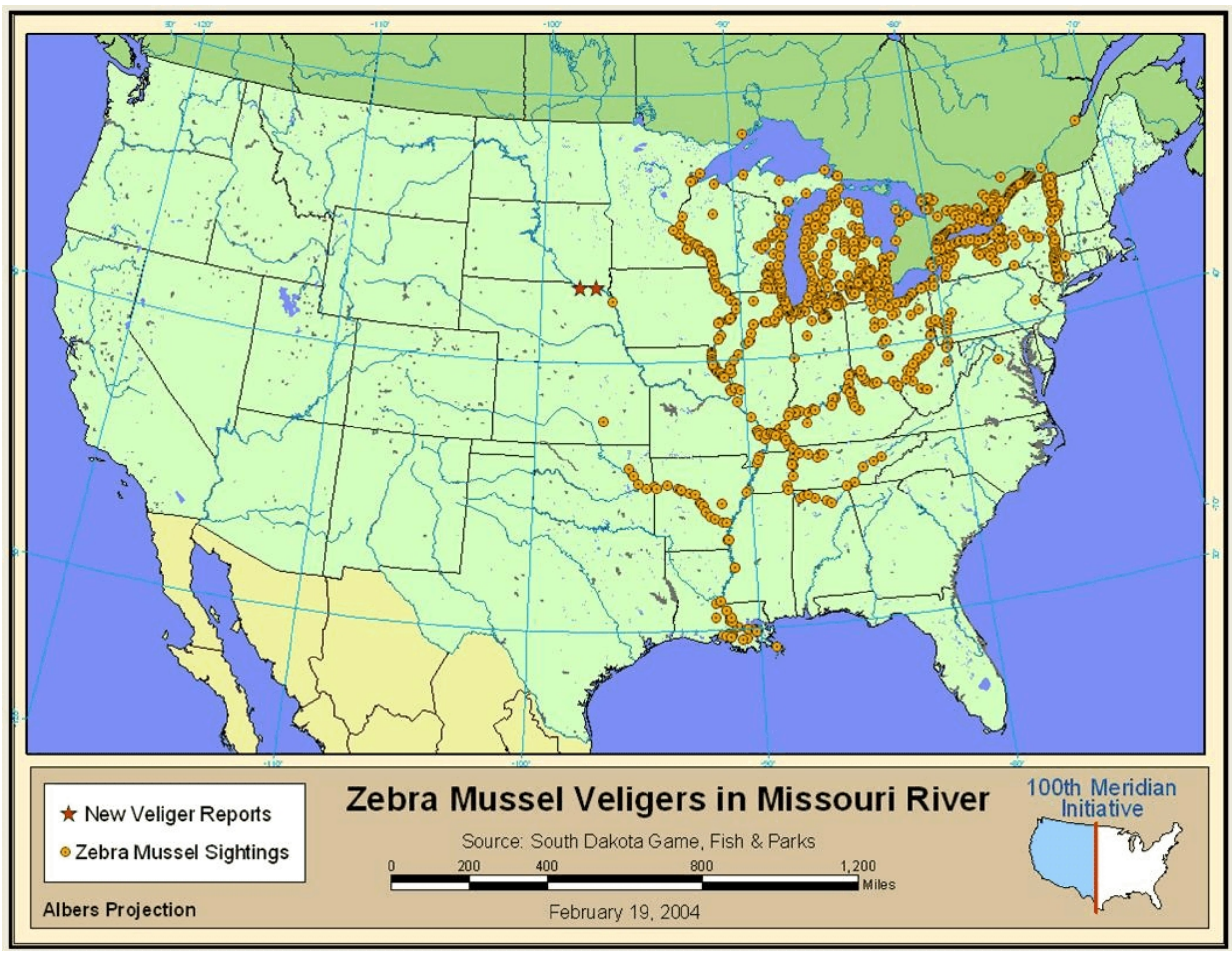

USGS
science for a changing world
Zebra Mussel Sightings Distribution
Dreissena polymorpha
Map produced by the U.S. Geological Survey, Gainesville, Florida, September 2002.

Figure 2. Zebra mussel distribution as of February, 2004 (including locations of observations of veligers).



[2]***Corbicula fluminea* Müller 1774**

Other scientific names appearing in the literature for this species: *Corbicula manilensis* (Philippi, 1844), *C. leana* Prime, *C. fluminalis*.

Common Name: Asian clam, propensity clam; *Corbicula*

Similar species Fingernail clam.

Size: Generally less than 50 mm in length.

Taxonomy and distinguishing features. Throughout their native range and in the US, Asian clams are highly variable in their external morphology (Sinclair, 1971a; Britton and Morton, 1986; Morton, 1987). At least part of this variability is believed to be influenced by environmental factors (Sinclair, 1971a). Asian clams introduced into the United States were initially identified as *C. fluminea* (Müller, 1774) (Lachner et al., 1970; Carlton, 1992) and *Corbicula manilensis* Philippi, 1844 (Sinclair, 1971a). In addition to these descriptions, Hillis and Patton (1982) presented morphological and electrophoretic data that suggested two species of *Corbicula* occurred in the United States, which is also the interpretation of genetic data published by McLeod (1986). In contrast to these interpretations, electrophoretic data presented by Morton (Morton in Blalock and Herod, 1999) and morphometric data presented by Britton and Morton (1986) suggests that all North American populations are really *C. fluminea*. Presently *C. fluminea* is used for all species collected in the US, although additional work into the taxonomic status of introduced populations may be necessary to resolve this dispute. (All North American records are C. fluminea with synonyms *C. fluminea* and *C. manilensis* and *C. leana*.)

Description. Adults may reach 50 to 65 mm in shell length (Hall, 1984), but individuals above 25 mm are typically uncommon (Gottfried and Osborne, 1982). Shells are rounded to slightly triangular and inflated; anterior and posterior margins rounded. Young shells have a purple stripe

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[2]Original materials prepared May, 2001 by A. M. Foster, Pam Fuller, and Amy Benson at the Center for Aquatic Resource Studies. The Center is part of the Biological Resources Division of the Geological Survey within the U.S. Department of the Interior. Additional materials incorporated from Illinois Natural History Survey and Windsor Aguirre (currently, State University of New York at Stony Brook) and Stuart G. Poss at the University of Southern Mississippi, 15 September 1999 and 6 October 1999. Updated, expanded, and edited, January, 2004.

on the umbo. Shell interiors are layered with polished, light purple nacre, and in adults the umbo is high, centrally located, and elevated above the hinge line. Shell color varies with the periostracum ranging from yellowish-brown to black. Dark-shelled members of the species are generally found in the southwestern United States, with periostracum bing a dark olive-green to black and a deep royal blue nacre. Light-colored *C. fluminea* have shells having yellow-green to light brown periostracum and white to light blue or light purple nacre (McMahon 1991). Regardless of color, the shell is ornamented by distinct, concentric sulcations, anterior and posterior lateral teeth with many fine serations. The shell has three cardinal teeth in each valve below the umbo, two on each side of the umbo in the right valve, one on each side in the left valve. The presence of serrated lateral teeth separate *Corbicula* from the fingernail-clams.

Life History and biology. The sexes are normally separate, however, hermaphrodites exist and are capable of self-fertilization. The spawning season lasts about 6 months starting in early summer. Fertilization takes place in the inner gills. The larval stages of the Asian clam differ significantly from the larval stages of other types of bivalves. In contrast to most bivalves, larval development occurs predominately inside the gill compartments of the female clam, and the development of internal body structures, including the foot and siphon, occurs at a slightly different time during development relative to the pattern characteristic of free-living veliger larvae. While larvae of Dreissenidae are completely pelagic (i.e., free-floating in the water column), other freshwater bivalves (e.g., Corbiculidea, Unionidae, Sphaeriidea) brood their larvae through early, pre-shell developmental stages in marsupial sacs on the gill.

The first larval stage called a trochophore (15-20 microns) develops on the inner gill. The second larval stage called the veliger (~0.2 mm) is also incubated on the gill, and a characteristic D-shaped shell begins to develop. At a shell length of approximately 1 mm, the juvenile is released from the gill and begins life as an adult on the bottom. At this free-living early adult stage, pigments and growth rings should be visible on the shell; maturity should be reached when shell length is 6-10 mm. Life span varies with habitat, but is generally 1-4 years, with a maximum life span of approximately 7 years (Hall, 1984). These mussels are filter-feeder primarily foraging on phytoplankton.

As adults, *Corbicula fluminea* is found in lotic and lentic habitats throughout its native range in southeastern Asia, and in the United States, the species has been most successful in well-oxygenated, clear waters (Belanger et al., 1985; Stites et al., 1995). The clams typically occur at high densities and have a relatively high growth rate (Stites et al., 1995). Densities of *C. fluminea* have been documented to occur by the thousands per square meter, often dominating the benthic community (Sickel 1986), with maximum densities reported to vary between 1000/m^2(Gotfried and Osborne, 1982; Stites et al., 1995) to 6000/ft^2 greater (Sinclair, 1971a; Sinclair, 1971b). Usually *C. fluminea* is more common and occurs at higher densities in stream pools than in stream runs (Blalock and Herod, 1999). Fine clean sand, clay, and coarse sand are preferred substrates, although this species may be found in lower numbers on most any substrate (Gottfried, and Osborne, 1982; Belanger et al., 1985; Blalock and Herod, 1999). Gottfried and Osborne (1982) reported density as lowest on bottoms composed of silty organic sediments. Factors that may affect population density and distribution of Asian clams include excessively high or low temperatures, salinity, drying, low pH, silt, hypoxia, pollution, bacterial, viral and parasitic infections, inter- and intraspecific competition, predators, and genetic changes (Evans et al. 1979; Sickel 1986).

Tolerance to reduced salinity and reduced water quality. Asian clams can tolerate salinities of up to 13ppt for short periods of time. If allowed to acclimate, they may tolerate salinities as high as 24ppt (King et al., 1986). Optimum is at lower salinities (Morton and Tong, 1985). In nature, Asian clams occur mostly in freshwaters, however, they have been reported from brackish and estuarine habitats, but are typically not as abudant in such habitats as in freshwaters (Carlton, 1992). In addition, Asian clams appear to be capable of tolerating polluted environments better than many native bivalves (Jenkinson, 1979).

Temperature Tolerance. This species appears to tolerate low temperatures well. Janech and Hunter (1995) reported a viable population surviving temperatures of 0-2˚C over winter in the Clinton River, Michigan (Janech and Hunter, 1984). Reproduction, on the other hand, is limited by low temperatures since velligers are typically released at temperatures of 16˚C or higher (Hall, 1984).

Reproduction and Fecundity. In North America, Asian clams breed from spring to fall. Reproductive activities are typically highest in the fall (Kraemer and Galloway, 1986). Asian clams are synchronous hermaphrodites and incubate their young within the inner demibranch of the ctenidium (branchial cavity). Self-fertilization may occur (Kraemer and Galloway, 1986). Sperm is ejected through the exhalent siphons (King et al., 1986). Egg cells are 120 to 170 µm in diameter just prior to fertilization (King et al., 1986). Trocophore larvae develop after 14 hours. Pediveligers are released from the parent in 4-5 days (King, 1979). Approximately between 320 (fall) and 387 (spring) pedivelligers are released daily per clam (Gottfried and Osborne, 1982;

Hall, 1984). Larval density has been reported to be as high as 1,000/ml (Sinclair, 1971b). When the pedivelligers are between 1.0 and 1.5mm in shell length, they attach to appropriate substrates with their byssus (Hall, 1984; Kraemer and Galloway, 1986). Larvae spawned in late spring and early summer may reach sexual maturity by the fall (Hall, 1984; King et al., 1986).

Trophic Interactions. Asian clams are consumed mainly by fish and crayfish. These clams have been found in the stomachs of black buffalo (*Ictiobus niger*; Minckley 1973); carp (*Cyprinus carpio*), channel catfish (*Ictalurus punctatus*), yellow bullhead (*Ameiurus natalis*), redear sunfish (*Lepomis microlophus*), largemouth bass (*Micropterus salmoides*), Mozambique tilapia (*Tilapia mossambica*; Minckley 1982); blue catfish (*Ictalurus furcatus*; M. Moser pers. comm. 1996); and spotted catfish (*Ameiurus serracanthus*; A. Foster pers. comm. 1996). In Florida, they have been reported as major prey items for the following species of fish: Redear sunfish, *Lepomis microlophus*, spotted bullhead, *Ameirus serracanthus* (Bass and Hitt, 1974), bluegill, *Lepomis macrochirus*, spotted sucker, *Minytrema melanops*, sturgeon, *Accipenser spp.*, channel catfish, *Ictalurus punctatus*, common carp, *Cyprinus carpio*, freshwater drum, *Aplodinotus grunniens*, smallmouth buffalo, *Ictiobus bubalus*, black buffalo, *I. niger*, and blue catfish, *Ictalurus furcatus* (McMahon, 1983).

Other predators of *Corbicula* include birds, raccoons, crayfish, and flatworms (Sickel 1986).

Habitat. Found in sandy and muddy bottomed streams and rivers, and in ponds and lakes of all sizes in silt, mud, sand, or gravel bottoms. Also, the species has also been recorded in man-made canals. *C. fluminea* can withstand habitats characterized by degraded water quality.

Native Range. The genus *Corbicula* is a freshwater species whose native range occupies temperate to tropical southern Asia west to the eastern Mediterranean (Russia, Thailand, Philippines, China, Hong Kong, Taiwan, Korea, and Japan) and Africa, except in the Sahara desert; southeast Asian islands south into central and eastern Australia (Morton 1986).

Nonindigenous Occurrences: The date and means of introduction of the Asian clam is not known. Generally, the introduction of this species is attributed to Chinese immigrants who used Asian clams as food (Sinclair, 1971a; Counts, 1981). The earliest verifiable record of this species in North America consists of three specimens found dead on the beach at Nanaimo, Vancouver Island, British Columbia in 1924 (USNM 363020; Counts, 1981). Asian clams are believed to have established a viable population on the west coast of the United States sometime prior to

1938 (Clench, 1970; Counts, 1981), perhaps established as early as the mid 1800's (Fox, 1969). Early records taken in the late 1930's and early 1940's exist for the Sacramento and San Joaquin River systems in California and the Columbia river system in Washington (Ingram, 1948). First documented in the US in California in 1938 where the species proliferated and spread by way of irrigation canals. A big migration east occurred when they were discovered in the Ohio River in 1957.

During this same period of "jump dispersal" eastward, the species rapidly invaded the Colorado, and spread throughout the Tennessee and Ohio River systems, spreading east along the Gulf states to the Florida panhandle by 1960, and to southern Florida by 1967 (Blalock and Herod, 1999). It was first reported for the Mobile River in 1962, where it was described as "abundant" (Hubricht, 1963). It was first reported for the Savannah River in 1972 (Stites et al., 1995) and reached Virginia that same year (Lachner et al., 1970). Transport on barges containing river gravel probably contributed to its rapid spread (Lachner, 1970; Sinclair, 1971a). Such gravel is often shipped great distances and maintains sufficient humidity to allow the clams to survive (Lachner et al., 1970). Other means of dispersal may have included transport on waterfowl, intentional releases by recreational aquarists, and intentional or accidental releases by fisherman who used this species as bait (Lachner et al., 1970; Sinclair, 1971a; Counts, 1981). Throughout the US, they can be found in many drainages except for the several states in the northern plains and the northeast. The Asian clam has long been recognized as a pest as far back as the 1950's in irrigation systems of California. *Corbicula* continues to expand its range into uninfested waters especially in the Midwest and Northeast. Asian clams have had one of the most rapid range expansions of any non-indigenous species in North America (Clench, 1970).

Status: Introduced, but widespread and common. *Corbicula fluminea* has become established in many states (see Figure 3).

Distribution. Asian clams naturally occur in southeast China, Korea, and in the Ussuri Basin, southeastern Russia (Lachner et al., 1970). Once translocated to North America, *C. fluminea* has flourished. The species is found in fresh waters throughout the United States including all five Gulf states and northern Mexico (Dundee, 1974; Carlton, 1992). Estuarine populations have been reported for the San Francisco Bay, California and Chesapeake Bay, Virginia, but none have yet been reported for the Gulf of Mexico ecosystem (Carlton, 1992). The species has been recorded in freshwater habits of the Gulf Coast states, however. In Alabama, besides the expanding populations from the west, *C. fluminea* may have been introduced into the Saugahatchee Creek from experimental ponds of the Auburn University Department of Fisheries and Applied Aquaculture in the summer of 1972 (Jenkinson, 1979). Howells (1992) noted the history of introduction of *C. fluminea* into Texas is largely unknown. However, the Asian clam has been reported as common in freshwaters of Texas (Howells, 1992).

Means of Introduction. The first collection of *C. fluminea* in the United States occurred in 1938 along the banks of the Columbia River near Knappton, Washington (Counts 1986). Since this first introduction, it is now found in at least 38 states and the District of Columbia. *Corbicula fluminea*. With human intervention a primary agent of dispersal, no large-scale geographic features function as dispersal barriers (Counts 1986; Isom 1986). Current methods of introduction include bait-bucket introductions (Counts 1986), accidental introductions associated with imported aquaculture species (Counts 1986), and intentional introductions by people who buy them as a food item in markets (Devick 1991). The only other significant dispersal agent is thought to be passive movement via water currents (Isom 1986); fish and birds are not considered to be significant distribution vectors (Counts 1986; Isom 1986).

Impact of Introduction: Because of their reproductive success and high infestation, this species has become a serious pest throughout the United States, especially in irrigation and drainage canals, as well as water distribution and industrial water use systems (Lachner et al., 1970; Sinclair, 1971a; Sinclair, 1971b; Mattice, 1977; Hall, 1984; Kraemer and Galloway, 1986; Stites et al., 1995). Given their high growth and production rates, concerns have been raised over the capacity that Asian clams have to alter trophic and nutrient dynamics of aquatic systems, and to displace native bivalves (Gottfried and Osborne, 1982; Stites et al., 1995).

The most prominent effect of the introduction of the Asian clam into the United States has been biofouling, especially of complex power plant and industrial water systems (Isom et al. 1986; Williams and McMahon 1986). It has also be documented to cause problems in irrigation canals and pipes (Prokopovich and Hebert 1965; Devick 1991) and drinking water supplies (Smith et al. 1979). It also alters benthic substrate (Sickel 1986), and competes with native species for limited resources (Devick 1991).

Over its native range the Asian clam is marketed fresh or dry for human consumption and as feed for domestic fowl (Lachner et al., 1970; Sinclair, 1971b), and in the United States the species has gained commercial value as fish bait (Lachner, 1970; Burch, 1978).

Control and management. The Asian clam is likely to spread in North America until it reaches its lower temperature tolerance. In closed environments, such as power plants, mechanical or chemical control methods can be employed to reduce or eliminate this species where problems occur. To eliminate the source of many introductions, navigation and dredging activities should be investigated.

References

Bass, D.G., Jr., and V.G. Hitt. 1974. Utilization of the Asiatic clam, *Corbicula manilensis*, by game fishes in Florida. Florida Game and Freshwater Fish Commission, Northeast Region, Lake City. 7pp.

Belanger, S.E., J.L. Farris, D.S. Cherry, and J. Cairns, Jr. 1985. Sediment preference of the freshwater Asiatic clam, *Corbicula fluminea*. The Nautilus 99(2-3):66-73.

Blalock, H.N., and J.J. Herod. 1999. A comparative study of stream habitat and substrate utilized by *Corbicula fluminea* in the New River, Florida. Florida Scientist 62:145-151.

Britton, J.C., and B. Morton. 1986. Polymorphism in *Corbicula fluminea*(Bivalvia: Corbiculoidea) from North America. Malacological Review 19:1-43.

Burch, B.L. 1978. Asian clam, *Corbicula* threatens Hawaii. The Nautilus 92(1):54-55.

Burch, J. Q. 1944. Checklist of west American mollusks. Minutes, Concological Club of southern California 38:18.

Carlton, J.T. 1992. Introduced marine and estuarine mollusks of North America: An end-of-the-20th-century perspective. Journal of Shellfish Research 11(2):489-505.

Clarke, A. H. 1981. *Corbicula fluminea* in Lake Erie. Nautilus 95:83-84.

Clench, W. J. 1971. *Corbicula manilensis* Philippi in Oklahoma. Nautilus 85:145.

Clench, W.J. 1970. *Corbicula manilensis* (Philippi) in lower Florida. Nautilus 84(1):36-37.

Counts, C. L. III. 1991. *Corbicula* (Bivalvia: Corbiculidae). Tryonia: Miscellaneous Publications of the Department of Malacology, No. 21. The Academy of Natural Sciences of Philadelphia. 134pp.

Counts, C. L., III. 1986. The zoogeography and history of the invasion of the United States by *Corbicula fluminea* (Bivalvia: Corbiculidae). American Malacological Bulletin, Special Edition No. 2:7-39.

Counts, C. L. III. 1985. *Corbicula fluminea* (Bivalvia: Corbiculidae) in the state of Washington in 1937, and in Utah in 1975. Nautilus 99:18-19.

Cummings, S. E. and J. A. Jones. 1978. Occurrence of *Corbicula manilensis* Philippi (sic) in the lower Minnesota River. Journal of the Minnesota Academy of Science 34:13-14.

Devick, W. S. 1991. Patterns of introductions of aquatic organisms to Hawaiian freshwater habitats. Pages 189-213 *in* New Directions in Research, Management and Conservation of Hawaiian Freshwater Stream Ecosystem. Proceedings Freshwater Stream Biology and Fisheries Management Symposium. Department of Land and Natural Resources, Division of Aquatic Resources, Honolulu, HI.

Diaz, R. J. 1974. Asiatic clam, Corbicula manilensis (Philippi) in the tidal James River, Virginia. Chesapeake Science 15:118-120.

Dressler, P. V. and R. L. Cory. 1980. The Asiatic clam, *Corbicula fluminea* (Müller), in the tidal Potomac River, Maryland. Estuaries 3:150-152.

Dundee, D.S. 1974. Catalog of introduced molluscs of eastern North America (north of Mexico). Sterkiana No. 55:1-37.

Dundee, D. S. and H. A. Dundee. 1958. Extension of known ranges of four Mollusca. Nautilus 72:51-53.

Eckblad, J. W. 1975. The Asiatic clam, *Corbicula*, in the upper Mississippi River. Nautilus 89:4.

Evans, L. P. Jr., C. E. Murphy, J. C. Britton and L. W. Newland. 1979. Salinity relationships in

Corbicula fluminea (Müller). Proceedings First International *Corbicula* Symposium. Texas Christian University Research Foundation, pp. 194-214.

Fetchner, F. K. 1962. *Corbicula fluminea* (Müller) from the Ohio River. Nautilus 75:126.

Fox, R.O. 1969. The *Corbicula* story: A Progress Report. Western Soc. Malacol. 11pp.

Fox, R. O. 1969. The *Corbicula* story: a progress report. Second Annual Meeting, Western Society of Malacologists. 11 pp.

Fox, R. O. 1970. The *Corbicula* story: chapter two. Third Annual Meeting, Western Society of Malacologists. 10 pp.

Fox, R. O. 1971. The *Corbicula* story: chapter three. Fourth Annual Meeting, Western Society of Malacologists. 5pp.

Fuller, S. L. H. and C. E. Powell. 1973. Range extension of *Corbicula manilensis* (Philippi) in the Atlantic drainage of the United States. Nautilus 87:59.

Gottfried, P.K., and J.A. Osborne. 1982. Distribution, abundance and size of *Corbicula manilensis* (Philippi) in a spring-fed central Florida stream. Florida Scientist 45(3):178-188.

Hall, J.J. 1984. Production of immature *Corbicula fluminea*(Bivalvia:Corbiculidae), in Lake Norman, North Carolina. The Nautilus 98(4):153-159.

Hanna, G. D. 1966. Introduced mollusks of western North America. California Academy of Sciences Occasional Papers 48:1-108.

Heard, W. H. 1966. Further records of *Corbicula fluminea* (Müller) in the southern United States. Nautilus 79:105-107.

Heard, W. H. 1964. *Corbicula fluminea* in Florida. Nautilus 77:105-107.

Hillis, D.M., and J.C. Patton. 1982. Morphological and electrophoretic evidence for two species of *Corbicula* (Bivalvia: Corbicularidae) in North America. American Midland Naturalist

108:74-80.

Hillis, D. M. and J. C. Patton. 1982. Morphological and electrophoretic evidence for two species of Corbicula (Bivalvia: Corbiculidae) in North America. American Midland Naturalist 108:74-80.

Howells, R.G. 1992. Annotated list of introduced non-native fishes, mollusks, crustaceans and aquatic plants in Texas. Texas Parks and Wildlife Management Data Series No. 78. Austin. 19 pp.

Hubricht, L. 1963. *Corbicula fluminea* in the Mobile River. Nautilus 77:31.

Hubricht, L. 1963. *Corbicula fluminea* in the Mobile River. Nautilus 77(1):31.

Ingram, W.M. 1948. The larger fresh-water clams of California, Oregon, and Washington. Journal of Entomology and Zoology 40(4):72-92.

Ingram, W.H. 1959. Asiatic clams as potential pests in California water supplies. Journal of the American Water Works Association 51:363-370.

Ingram, W. H. 1948. The larger freshwater clams of California, Oregon and Washington. Journal of Entomology and Zoology 40:72-92.

Isom, B. G., C. F. Bowman, J. T. Johnson, E. B. Rodgers. 1986. Controlling *Corbicula* (Asiatic clams) in complex power plant and industrial water systems. American Malacological Bulletin, Special Edition 2:95-98.

Isom, B.G. 1986. Historical review of Asiatic clam (*Corbicula*) invasion and biofouling of waters and industries in the Americas. American Malacological Bulletin, Special Edition No. 2:1-5.

Janech, M.G., and R.D. Hunter. 1995. Corbicula fluminea in a Michigan river: Implications for low temperature tolerance. Malacological Review 28:119-124.

Jenkinson, J.J. 1979. The occurrence and spread of *Corbicula manilensis*in east-central Alabama. Nautilus 94(4):149-153.

King, C.A., C.J. Langdon, and C.L. Counts, III. 1986. Spawning and early development of

Corbicula fluminea (Bivalvia: Corbicularidae) in laboratory culture. American Malacological Bulletin 4(1):81-88.

Kraemer, L.R., and M.L. Galloway. 1986. Larval development of *Corbicula fluminea* (Muller) (Bivalvia: Corbiculacea): An appraisal of its heterochrony. American Malacological Bulletin 4(1):61-79.

Lachner, E.A., C.R. Robins, and W.R. Courtenay, Jr. 1970. Exotic fishes and other aquatic organisms introduced into North America. Smithsonian Contributions to Zoology No. 59. 29 pp.

Livo, L. J. 1996. Colorado Outdoors 45:17-19.

Mackie, G. L. and D. G. Huggins. 1983. Sphaeriacean clams of Kansas. Technical Publications of the State Biological Survey of Kansas, University of Kansas, Lawrence. 92 pp.

Mattice, J.S. 1977. Interactions of *Corbicula sp.* with power plants. Pages 119-138 in J.C. Britton (ed.), Proceedings of the First International Corbicula Symposium. Texas Christian University Research Foundation. Fort Worth, Texas. 313 pp.

McLeod, M. J. 1986. Electrophoretic variation in North American *Corbicula*. American Malacological Bulletin, Special Edition 2:125-132.

McMahon, R. 1991. Mollusca: Bivalvia, pages 315-399 in Thorp, J. H. and A. P. Covich. (eds.) Ecology and Classification of North American Freshwater Invertebrates. Academic Press, Inc., New York. 911pp.

McMahon, R.F. 1983. Ecology of an invasive bivalve, *Corbiclua*. Pages 505-561 in W.D. Russell-Hunter (ed.) The Mollusca. Vol 6. Ecology. Academic Press. San Diego.

Metcalf, A. L. 1966. *Corbicula manilensis* in the Mesilla Valley of Texas and New Mexico. Nautilus 80:16-20.

Miller, A.C., and B.S. Payne. 1994. Co-occurrence of native freshwater mussels (Unionidae) and the non-indigenous *Corbicula fluminea* at two stable shoals in the Ohio River, USA. Malacological Review 27:87-97.

Minckley, W. L. 1973. Fishes of Arizona. Arizona Game and Fish Department, Phoenix

Minckley, W. L. 1982. Trophic interrelations among introduced fishes in the lower Colorado River, southwestern United States. California Fish and Game 68:78-89.

Morton, B., and K.Y. Tong. 1985. The salinity tolerance of *Corbicula fluminea* (Bivalvia:Corbiculoidea) from Hong Kong. Malacological Review 18:91-95.

Morton, B. 1979. *Corbicula* in Asia. Pages 15-38 in Proceedings, First International *Corbicula* Symposium, Texas Christian University, Fort Worth, Texas. 13-15 October, 1977. 313 pp.

Morton, B. 1986. *Corbicula* in Asia - an updated synthesis. American Malacological Bulletin, Special Edition No. 2:113-124.

Morton, B. 1987. Polymorphism in *Corbicula fluminea*(Bivalvia:Corbiculoidea) from Hong Kong. Malacological Review 20:105-127.

Peyton, M. M. and J. L. Maher. 1995. A survey of mussels (Mollusca: Bivalvia) in the Platte River system and associated irrigation and hydropower canal and lake systems west of Overton, Nebraska. Transactions of the Nebraska Academy of Sciences 22:43-48.

Pojeta, J. 1964. Notes on the extension of the known range of the Asiatic clam *Corbicula fluminea* (Müller) in Ohio River. Ohio Journal of Science 64:428.

Prokopovich, N. P. and D. J. Hebert. 1965. Sedimentation in the Delta-Mendota Canal. Journal of the American Water Works Association 57:375-382.

Raeihle, D. 1983. *Corbicula fluminea* (Müller, 1774) in Massapequa Lake, Long Island, New York. New York Shell Club Notes 28:9-10.

Sickel, J. B. 1986. *Corbicula* population mortalities: factors influencing population control. American Malacological Bulletin, Special Edition 2:89-94.

Sickel, J. B. 1973. New record of *Corbicula manilensis* (Philippi) in the southern Atlantic slope region of Georgia. Nautilus 87:11-12.

Sinclair, R.M. 1971b. Annotated bibliography on the exotic bivalve *Corbicula*in North America, 1900-1971. Sterkiana No. 43:11-18.

Sinclair, R.M. 1971a. Corbicula variation and Dreissena parallels. The Biologist 53(3):153-159.

Sinclair, R. M. and B. G. Isom. 1961. A preliminary report on the introduced Asiatic clam *Corbicula* in Tennessee. Tennessee Stream Pollution Control Board, Tennessee Department of Public Health. 31 pp.

Smith, A. L., A. Mula, J. P. Farkas and D. O. Bassett. 1979. Clams - a growing threat to inplant water systems. Plant Engineering 1979:165-167.

Stein, C. B. 1962. An extension of the known range of the Asiatic clam, *Corbicula fluminea* (Müller), in the Ohio and Mississippi Rivers. Ohio Journal of Science 62:326-327.

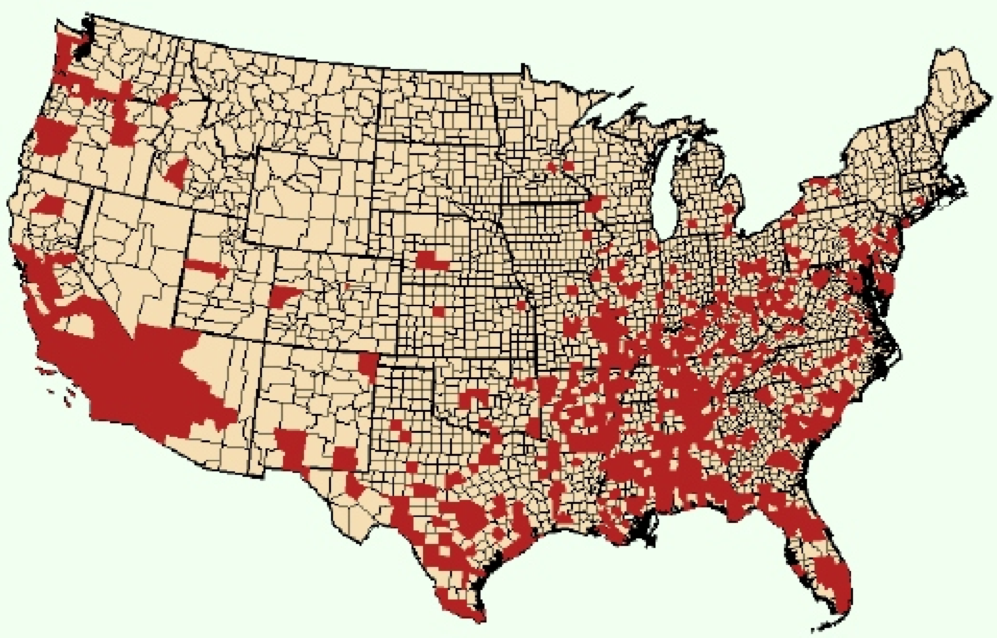
Stites, D.L., A.C. Benke, and D.M. Gillespie. 1995. Population dynamics, growth, and production of the Asiatic clam, *Corbicula fluminea*, in a blackwater river. Canadian Journal of Fisheries and Aquatic Sciences 52:425-437.

Stotts, V. D., K. D'Loughy, and D. B. Stotts. 1977. Waterfowl habitat. Maryland Department of Natural Resources Wildlife Administration Report 77-2. 6 pp.

Thomas, N. A. and K. M. MacKenthum. 1964. Asiatic clam infestation at Charleston, West Virginia. Nautilus 78:28-29.

Williams, C. J. and R. F. McMahon. 1986. Power station entrainment of Corbicula fluminea (Müller) in relation to population dynamics, reproductive cycle and biotic and abiotic variables. American Malacological Bulletin, Special Edition 2:99-111.

Figure 3. Current distribution map and chronology of invasion. Based largely on the exhaustive taxonomic work of Counts (1981), the current distribution of *C. fluminea* is illustrated below (county records) and chronology summarized in legend..



Corbicula distribution by county. From Counts (1986) and others. March 1999.

Chronology of invasion. Since its documented introduction to the United States in 1938, *C. fluminea* has spread into many of the major waterways. The following location information briefly outlines where it is presently found. The [date: author publication date] format associated with each state identifies the first collection or record of *C. fluminea* in that state. The Asian clam has become established in the following states: Alabama [1962: Hubricht 1963] widespread (Counts 1991); Arizona [1958: Dundee and Dundee 1958] in the Aqua Fria, Colorado, Gila, Salt, and Verde rivers; Lake Martinez; and in several irrigation systems in Maricopa County (Counts 1991); Arkansas [1970: Fox 1970] widespread (Counts 1991); California [1945: Hanna 1966] in the Sacramento and San Joaquin drainages; Santa Barbara County south to San Diego County and west to the Salton Sea (Counts 1991); Colorado [1995: Livo 1996] in a northwestern reservoir; Connecticut [1990: Morgan, pers. comm.] in the Connecticut River; Delaware [1986: Counts 1986] in the Delaware River in New Castle County; the Nanticoke River in Sussex County; and the Nanticoke Wildlife Refuge (Counts 1991); District of Columbia [1979:Dressler and Cory 1980] in the Potomac River; Florida [1964: Heard 1964] widespread (Counts 1991; J. D.

Williams pers. comm. 1996); Georgia [1971: Sickel 1973] widespread (Counts 1991); Hawai`i [1982: Devick 1991] on the islands of O`ahu, Kaua`i, Maui, and Hawai`i; Idaho [1959: Ingram 1959] in the Snake River on the Idaho-Washington state line; Illinois [1962: Fetchner 1962] in the Illinois River south to the state line (Counts 1991); Indiana [1962: Fox 1969] in the White, lower Wabash, and Blue river drainages; Big Indian and Indian Creeks; and the Ohio River in Clark and Posey Counties (Counts 1991); Iowa [1974: Eckblad 1975] in the Mississippi River near Lansing; and the Cedar River in Linn County (Counts 1991); Kansas [1983: Mackie and Huggins 1983] in Perry Reservoir on the Delaware River; the Kansas River drainage; the North Fork of the Ninnescah River; Wilson Reservoir on the Saline River; and Cedar Bluff Reservoir on the Smoky Hill River (Counts 1991); Kentucky [1957: Sinclair and Isom 1961] widespread (Counts 1991); Louisiana [1961: Stein 1962] in the Pearl, Atchafalaya, Mississippi, and upper Red drainages (Counts 1991); Maryland [1975: Stotts et al. 1977] in the Choptank River near Goldsboro; Nassawango Creek near Snow Hill; the Susquehanna River below Conowingo Dam; the Wicomico River at Salisbury; the Potomac River in Charles, Prince Georges, and Montgomery Counties; Chesapeake Bay at Havre-de-Grace, and near the mouth of the Susquehanna River (Counts 1991); Michigan [1981: Clarke 1981] in Lake Michigan at the J. H. Campbell Power Plant; and Lake Erie at Detroit Beach, Sterling State Park and Bolles Harbor (Counts 1991); Minnesota [1975: Cummings and Jones 1978] in the Minnesota River near Burnsville; Mississippi [1963: Heard 1966] widespread (Counts 1991); Missouri [1969: Fox 1969] in the lower Missouri River drainage south to the state line; Nebraska [1991: Peyton and Maher 1995] in the Platte River in Lincoln and Dawson Counties; Nevada [1959: Ingram 1959] in Lake Meade (Counts 1991); New Jersey [1973: Fuller and Powell 1973] in the Raritan River in Middlesex and Somerset Counties; and the Delaware River near Newbold Island, Wright Point, and Trenton (Counts 1991); New Mexico [1966: Metcalf 1966] in Nemexas-West Drain in Dona Ana Co.; the Pecos River impoundment at Riverside Drive in Carlsbad; and the Rio Grande River from Caballo and Elephant Butte reservoirs, south to Percha Dam (Counts 1991); New York [1983: Raeihle 1983] in Massapequa Lake on Long Island; North Carolina [1970: Fox 1971] in the Cape Fear, Catawba, Chowan, Eden, Little, Meherrin, Neuse, Roanoke, Rocky, Tar, Uhwarrie, and Waccamaw rivers; and Richardsons Creek (Counts 1991); Ohio [1962: Pojeta 1964] in the Muskingum, upper Scioto, and upper Great Miami drainages; and the lower Hocking River (Counts 1991); Oklahoma [1969: Clench 1971] in the Arkansas River from Cherokee to Wagoner Counties; the Little River near Goodwater; Lake Texoma on the Red River; Lake Overholser; Lake Thunderbird; and Caddo Creek in Carter County (Counts 1991); Oregon [1948: Ingram 1948] in the Columbia drainage; the John Day River; the Smith River near Scottsburg; and at the mouth of the Siuslaw and Willamette rivers (Counts 1991); Pennsylvania [1973: Fuller and Powell

1973] in the Ohio and Delaware rivers; the Beaver River in Beaver County; the Monongahela River at Lock and Dam Number 8; and the Schuykill River at the Limerick Power Station and Fairmount Dam (Counts 1991); South Carolina [1972: Fuller and Powell 1973] in the Savannah, Cooper, Santee, Pee Dee, Little Pee Dee, Edisto, Waccamaw, and Salkahatchie rivers; the intracoastal waterway; and several industrial facilities in Aiken and Pickens counties (Counts 1991); Tennessee [1959: Sinclair and Isom 1961] in the Tennessee drainage (Counts 1991); Texas [1964: Metcalf 1966] in the Angelina, Colorado, Rio Grande, Guadalupe, San Antonio, San Jacinto, Sabine, Red, White, and Brazos drainages; the Clear and West Forks of the Trinity River (Counts 1991); Utah [1975: Counts 1985] in Sevier Reservoir; Virginia [1968: Diaz 1974] in the Appomattox, Clinch, Potomac, James, and New rivers; Lake Anna; the Chowan River at the mouths of the the Blackwater and Nottoway rivers; and the Chickahominy River at Lanexa; (Counts 1991); Washington [1938: Burch 1944] in the Columbia, Snake, Chehalis, and Willapa rivers; Hood Canal in Jefferson County; and Aberdeen Lake in Grays Harbor Lake County (Counts 1986, 1991); West Virginia [1964: Thomas and MacKenthum 1964] in the Elk and Kanawha drainages (Counts 1991); Wisconsin [1977: Cummings and Jones 1978] in the Mississippi River near Prairie du Chien and La Cross; and the St. Croix River near Hudson (Counts 1991).

[3]***Potamopyrgus antipodarum*** (J. E. Gray, 1853)

Common Name: New Zealand mudsnail

Size: Approximately 5mm in length, ranging to 12mm. The shell usually displays right-handed coiling with 7 to 8 whorls.

Native Distribution. The New Zealand mudsnail (*Potamopyrgus antipodarum*) is a small aquatic snail, native to freshwater lakes and streams of New Zealand and small islands immediately adjacent to New Zealand.

Life History. Mudsnail populations consist mostly of asexually reproducing females that are born with developing embryos in their reproductive system. New Zealand mudsnails are very small, operculate gastropods, and have a shell that varies from gray and dark brown to light brown in color.

This species can be found in all types of aquatic habitats from eutrophic mud bottom ponds to clear rocky streams. It can tolerate a wide range of water temperatures (except freezing), salinity, and turbidity in clean as well as degraded waters. They feed on dead and dying plant and animal material, algae, and bacteria. Mudsnail densities of over one-half million per meter square in western streams are a cause for concern. Because the West is known for abundant trout and productive fishing spots, there is concern that the mudsnails will impact the food chain for native trout and the physical characteristics of the streams themselves

Nonindigenous Occurrences: Outside New Zealand, the snail has relocated throughout the world, including Europe, Asia, and North America. The species has become naturalized in Australia and Europe. Movements of this aquatic snail to North America were likely associated with ship ballast in the Great Lakes or in the water of live gamefish shipped from infested waters to western rivers in the United States. New Zealand mudsnails were first discovered in the middle portion of the Snake River in Idaho in 1987, and currently occupy a disjunct distribution throughout North America (Figure 4). Since collection its collection in Idaho in the late 1980's, the species has spread to waters of Montana and Wyoming, including the waters of Yellowstone

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[3]Original material prepared by staff at USGS/BRD/FISC, and Dan Gustafson and colleagues at Montana State University. Expanded, updated, and edited January, 2004.

National Park, as well as California, Arizona, Oregon, and Utah. New Zealand mudsnails have also been collected in Oregon near the mouth of the Columbia River and in a small lake in northwestern area of the state. Populations have been discovered in the Owens River and Central Valley of California in 2001 and 2002, and in the Colorado River in Arizona and the Green River in Utah in 2002. See Figure 4 for current distribution in western US and throughout the Missouri River watershed (HUC10). Populations in the eastern US occur in Great Lakes where a populations were discovered in the early 1990's in southwestern and northeastern Lake Ontario and the Welland Canal in Canada, and in 2003 New Zealand mudsnails were collected from Ontario's Thunder Bay immediately off Lake Superior.

Methods of Control: The species' parthenogenicity and its tolerance of a broad range of ecological factors make the possibility of further spread highly likely. As long as not completely desiccated, New Zealand mudsnails can withstand short periods out of the water. Precautionary measures should include education of the public to decontaminate fishing and sporting equipment so as not to spread existing populations or start new ones. Regulations on commercial shipping of this species are in effect. While the species supports a number of parasites in its native range, none have been found in any of the North American populations examined.

[4]References

Adam, W.1942. Sur la repartition et la biologie de Hydrobia jenkinsi (Smith) en Belgique. Bulletin du Musee royal d'histoire naturell de Belgique 18: 1-18.

Allen, K. R. 1951. The Horokiwi Stream. A study of a trout population. Bulletin of New Zealand Marine Department of Fisheries 10A: 1-70.

Bailey, R. C. 1988. Correlations between species richness and exposure: Freshwater molluscs and macrophytes. Hydrobiologia 162: 183-191.

Barnese, L. E., R. L. Lowe, and R. D. Hunter 1990. Comparative grazing efficiency of Pulmonate and Prosobranch snails. Journal of the North American Benthological Society 9(1): 35-44.

[4]*Potomopyrgus antipodarum* bibliography originally Dave Richards and Billie Kerans, Montana State University; last accessed November, 2004).

Berner, L. 1963. Sur l'invasion de la France par Potamopyrgus jenkinsi. Arch. Moll. 92: 19-29.

Bick, A. a. M. L. Z. 1994. The distribution of Hydrobiids and the effect of sediment characteristics on the population dynamics of Hyrdobia ventrosa in a coastal region of southern Baltic. Int. Revue ges. Hydrobiol. 79(3): 325-336.

Boettger, C. 1953. Die Herkunft und verwandtschaftsbeziehungen der wasserschnecke Potamopyrgus jenkinsi, nebst eine Anaabe uber ihr Auftreten in Mediterrangebiet. Arch. Moll. 80: 57-84.

Bondesen, P. a. E. W. K. 1949. Hydrobia (Potamopyrgus) jenkinsi (Smith) in Denmark illustrated by its ecology. Oikos 1(2): 252-281.

Bowler, P. A. 1991. The rapid spread of the freshwater Hydrobiid snail Potamopyrgus antipodarum (Gray) in the Middle Snake River, Southern Idaho. Proceedings of the Desert Fishes Council.21:173-182

Bowler, P. A. and T. J. Frest .1992. The non-native snail fauna of the Middle Snake River, Southern Idaho. Proceedings of the Desert Fishes Council, New Mexico.

Bowler, P. A. and T. J. Frest. 1996. The advancing distribution of the New Zealand mud snail, Potamopyrgus antipodarum (Gray), in North America. Contributed paper. Am. Malacol. Union, 62nd annual meeting, Chicago, IL.

Boycott, A. C. 1919. Parthenogenesis in Paludestrina jenkinsi. Ibidem 16: 54.

Boycott, A. C. 1929. The inheritance of ornamentation in var. aculeata of Hydrobia jenkinsi Sm. Proc. Mal. Soc. London 18: 230.

Boycott, A. C.1936. The habitats of freshwater mollusca in Britain. Journal of Animal Ecology 5: 111-186.

Boycott, A. C.1947. Where is the male of Paludestrina jenkinsi. Journal of Conch. 15: 216.

Broekhuizen, N., D. Miller, and S. Parker (2001). Fine sediment effects on feeding and growth in the invertebrate grazers Potamopyrgus antipodarum (Gastropoda, Hydrobiidae) and Deleatidium sp. (Ephemeroptera, Leptophlebiidae). Hydrobiologia 457(1-3): 125-132.

Bronmark, C. (1988). Effects of vertebrate predation on freshwater gastropods: an enclosure experiment. Hydrobiologia 169: 363-370.

Burnet, A. M. R. A study of the inter-relation between eels and trout, the invertebrate fauna and the feeding habits of the fish. Source ???

Calow, P. and L. J. Calow. 1975. Cellulose activity and niche separation in freshwater gastropods. Nature 255: 478-480.

Cejka, T. 1994. First record of the New Zealand mollusc Potamopyrgus antipodarum (Gray 1843), (Gastropoda, Hydrobiidae) from the Slovak section of the Dunaj River. Biologia, Bratislava 49(5): 657-658.

Cogerino, L., B. Cellot, et al. 1995. Microhabitat diversity and associated macroinvertebrates in aquatic banks of a large European river. Hydrobiologia 304: 103-115.

Cohen, A. N. and J. T. Carlton.1998. Accelerating invasion rate in a highly invaded estuary. Science Washington D. C. 279 (5350) 555-558.

Cotton, B. C. 1942. Some Australian freshwater Gastropoda. Transactions of the Royal Society of Australia. 66: 75-82.

Crozet, B.1985. Influence of the sewage treatment plant effluents on the structure of the benthic communities of Lake of Geneva. Verh. Internat. Verein. Limnol. 22: 2327-2331.

Crozet, B., Pedroli, J. C., and C. Vaucher. 1980. Premieres observations Potamopyrgus jekinsi (Smith)(Mollusca, Hydrobiidae) en Suisse romande. Revue suisse Zoologie 87: 807-811.

Cuellar, O.1994. Biogeography of parthenogenetic animals. Compte Rendu des Seances de la Societe de Biogeographie 70: 1-13.

Cunha, M. R. a. M. H. M.1995. Macrobenthos of Potamogeton and Myriophillum beds in the upper reaches of Canal De Mira (Ria de Aveiro, New Portugal): Community structure and environmental factors. Netherlands Journal of Aquatic Ecology 29(3-4): 377-390.

Cyr, H. a. J. A. D.1988. The abundance of phytophilou invertebrates on different species of submerged macrophytes. Freshwater Biology 20: 365-374.

Dahl, A. a. L. B. W.1993. Life-history and growth of the prosobranch snail Potamopyrgus jenkinsi in Lake Esrom, Denmark. Verh. Internat., Verein. Limnol. 25: 582-586.

Dahl, A. and L. B. Winther.1993. Life-history and growth of the prosobranch snail Potamopyrgus jenkinsi in Lake Esrom, Denmark. Verh. Internat. Verein. Limnol. 25: 582-586.

Dean, J. D. (1904-06). Fish and their relation to Paludestrina jenkinsi. Journal of Conch. 11: 15.

Desert Fishes Council.1992. Relative to the proposed listing of five species of aquatic snails in Idaho as endangered. Proceedings of the Desert Fishes Council. Vol. 21 and 22: 72. 1990 and 1991 Annual Symposia. Ed. Dean A. Hendrickson. Bishop, California.

Dixon, M. and J. W. Carter.1999. Riparian vegetation along the Middle Snake River, Idaho: Zonation, geographical trends, and historical changes. Great Basin Naturalist. 59 (1) 18-34.

Doby, J. M., A. Charaud, et al.1965. Extension en Clorse du mollusque gastropode Potamopyrgus jenkinsi (Smith, 1889) (Hydrobiidae). Bulletin du Museum d'hsitoire naturelle, Paris 37: 833-843.

Dorgelo, J. 1987. Density fluctuations in populations (1982-1986) and biological observations of Potamopyrgus jenkinsi in two trophically differing lakes. Hydrobiological Bulletin 21(1): 95-110.

Dorgelo, J. 1988. Growth in a freshwater snail under laboratory conditions in relation to eutrophication. Hydrobiologia 157: 125-127.

Dorgelo, J.1991. Growth, food and respiration in the prosobranch snail Potamopyrgus jenkinsi (E. A. Smith)(Hydrobiidae, Molluska). Verh. int. Ver. Limnol. 24: 2947-2953.

Dorgelo, J., H. Meester, and C. van Velzen. 1995. Effects of diet and heavy metals on growth rate and fertility in the deposit-feeding snail Potamopyrgus jenkinsi (Smith) (Gastropoda: Hydrobiidae). Hydrobiologia 316: 199-210.

Duncan, A. 1966. The oxygen consumption of Potamopyrgus jenkinsi (Smith) (Prosobranchiata) in different temperatures and salinities. Verh. Internat. Verein. Limnol. 16: 1739-1751.

Duncan, A. and R. Z. Klekowski. 1967. The influence of salinity on the survival, respiratory rate and heart beat of young Potamopyrgus jenkinsi (Smith) Prosobranchiata. Comp. Biochem. Physiol. 22: 495-505.

Dussart, G. B. J. 1976. The ecology of freshwater molluscs in North West England in relation to water chemistry. Journal of Molluscan Studies 42: 181-198.

Dussart, G. B. J.1987. Effects of water flow on the detachment of some aquatic pulmonate gastropods. American Malacological Bulletin 5(1): 65-72.

Dybdahl, M. F. and C. M. Lively. 1992. Endangered and threatened wildlife and plants: determination of endangered or threatened status for five aquatic snails in south central Idaho. Vol. 57, No. 240. Fontaine, T. D. and D. J. Stewart.

Dybdahl, M. F. and C. M. Lively. 1992. Exploring the effects of multiple management objectives and exotic species on Great Lakes food webs and contaminant dynamics. En. Management. 16(2):225-229.

Dybdahl, M. F. and C. M. Lively. 1995. Host-parasite interactions: infection of common clones in natural populations of a freshwater snail. Proceedings of the Royal Society of London 260: 99-103.

Dybdahl, M. F. and C. M. Lively. 1995. Diverse, endemic and polyphyletic clones in mixed populations of a freshwater snail Potamopyrgus antipodarum. Journal of Evolutionary Biology. 8(3): 385-398.

Dybdahl, M. F. and C. M. Lively. 1996. The geography of coevolution: comparative population structures for a snail and its trematode parasite. Evolution 50(6): 2264-2275.

Dybdahl, M. F. and C. M. Lively. 1997. Genotypic variation of an exotic freshwater snail in the Snake and Madison Rivers (USA) populations. Abstract. Society for Conservation Biology Annual Meeting. Victoria, British Columbia.

Dybdahl, M. F. and C. M. Lively. 1998. Host parasite coevolution: evidence for rare advantage and time-lagged selection in a natural population. Evolution. 52: 1057-1066.

Falniowski, A. 1995. Can poorly understood new characters support a poorly understood phylogeny? Shell-structure data in Hydrobiid systematics (Mollusca: Gastropoda: Prosobranchia: Hydrobiidae). Journal of Zoological and Systematic Evolutionary Research 33: 133-144.

Fenchel, T. 1975. Character displacement and coexistence in mud snails (Hyrobiidae). Oecologia 20: 19-32.

Fenchel, T. 1975. Factors determining the distribution patterns of mud snails (Hydrobiidae). Oecologia 20: 1-17.

Fenchel, T. 1976. Evidence for exploitative interspecific competition in mud snails (Hydrobiidae). Oikos 27: 367-376.

Fish, J. D. and S. Fish. 1981. The early life-cycle stages of Hydrobia verntrosa and Hydrobia neglecta with observations on Potamopyrgus jenkinsi. Journal of Mollusc. Stud. 47: 89-98.

Foltz, D. W., H. Ochman, J. S. Jones, and R. K. Selander. 1985. Genetic heterogeneity within and among morphological types of the parthenogenetic snail Potamopyrgus jenkinsi. ?: 242-245.

Forbes, V. E. 1990. The role of sediment type in growth and fecundity of mud snails (Hydrobiidae). Oecologia 83: 53-61.

Forsyth, D. J. 1978. Benthic macroinvertebrates of Lake Taupo. N. Z. J. Mar. Fresh. Res. 15(1): 41-46.

Fox, J. A., M. F. Dybdahl, J. Jokela, nd C. M. Lively. 1996. Genetic structure of coexisting sexual and clonal subpopulations in a freshwater snail (Potamopyrgus antipodarum). Evolution 50(4): 1541-1548.

Fox, G. A., Allan, L. J., Weseloh, D. V., and P. Mineau (???). The diet of herring gulls during the nesting period in Canadian waters of the Great Lakes. Canadian J. Zool. 68(6): 1075-105.

Frenzel, V. P. 1979. Biology and population dynamics of Potamopyrgus jenkinsi (Smith) (Gastropoda: Prosobranchia) in the littoral of Lake Constance. Archives of Hydrobiologie 85(4): 448-464.

Frest, T.J. and P.A. Bowler.1993. A preliminary checklist of the aquatic and terrestrial mollusks of the Middle Snake River sub-basin. Proceedings of the Desert Fishes Council 24: 53-58.

Fretter, V. and A. Graham. 1978. The prosobranch molluscs of Britain and Denmark. Part 3. The Journal of Molluscan Studies. Supplement 5: 1-152.

Fromming, E. 1956. Biologie der mitteleuropaschen Susswasserschnecken. Duncker and Humblot Berlin.

Gallardo, A., Prenda, J., and A. Pujante. 1994. Influence of some environmental factors on the freshwater macroinvertebrate distribution in two adjacent river basins under Mediterranean climate: II. Mollusks. Archiv. fuer. Hydrobiologie. 131(4): 449-463.

Gerard, C. 1997. Importance du parasitisme dans la communaute de Gasteropodes de L'Etang de Combourg (Bretagne, France). Parasite 4: 49-54.

Golding, L. A., M. H. Timperley, and C. W. Evans. 1997. Non-lethal responses of the freshwater snail Potamopyrgus antipodarum to dissolved arsenic. Environmental Monitoring and Assessment 47: 239-254.

Gonzalez-Solis, J. a. X. R. 1996. Succession and secondary production of gastropods in the Ebro Delta ricefields. Hydrobiologia 337: 85-92.

Graynoth, E., Bennett, L. C., and J. C. Pollard. 1986. Diet of landlocked sockeye salmon (Oncohynchuys nerka) and trout in the Waitaki lakes, New Zealand. N. Z. J. Mar. Fresh. Res. 20: 537-549.

Harding, J. S. 1992. Discontinuities in the distribution of invertebrates in impounded South Island

Rivers, New Zealand. Regulated rivers Research and Mangement. 7(4):327-335.

Harman, W. N. 1974 in C. W. Hart and S. L. H. Fuller (eds.). Pollution Ecology of Freshwater Invertebrates: Snails (Mollusca: Gastropoda). Academic Press, New York. pp. 275-312.

Hauser, L., G. R. Carvalho, et al. 1992. Clonal structure of the introduced freshwater snail Potamopyrgus antipodarum (Prosobranchia: Hydrobiidae), as revealed by DNA fingerprinting. Proceedings of the Royal Society of Londen Series B 249: 19-25.

Hawkins, C. P. a. J. K. F. 1987. Are snails important competitors in stream ecosystems? Oikos 49: 209-220.

Haynes, A. and B. J. R. Taylor. 1984. Food finding and food preference in Potamopyrgus jenkinsi (E. A. Smith) (Gastropoda: Prosobranchia). Archives Hydrobiology 100(4): 479-491.

Haynes, A., B. J. R. Taylor, and M. E. Varley. 1985. The influence of the mobility of Potamopyrgus jenkinsi (Smith, E. A.) (Prosobranchia: Hydrobiidae) on its spread. Archives of Hydrobiologie 103(4): 497-508.

Haynes, A. 1990. The numbers of freshwater Gastropods on Pacific islands and the theory of Island Biogeography. Malacologia 31(2): 237-248.

Heywood, J. and R. W. Edwards. 1962. Some aspects of the ecology of Potamopyrgus jenkinsi (Smith). Journal of Animal Ecology 31: 239-250.

Hickey, C. W. a. M. L. V. 1994. Toxicity of ammonia to nine native New Zealand freshwater invertebrate species. Arhcives of Environmental Contamination and Toxicoligy 26: 292-298.

Holomuzki, D. J. and B. J. F. Biggs. 1998. Distributional responses to flow disturbance by a stream-dwelling snail. Bull. N. A. Benthological Soc. (Abstract) 15: 100.

Hopkins, C. L. 1976. Estimate of biological production in some stream invertebrates. N. Z. J. Mar. Fresh. Res. 10: 629-640.

Howard, R. S. and C. M. Lively. 1994. Parasitism, mutation accumulation and the maintenance of sex. Nature 367: 554-557.

Hubendick, B. 1950. The effectiveness of passive dispersal in Hydrobia jenkinsi. Zoologia Bidrag (Uppsala) 28: 493-504.

Hughes, R. N. 1996. Evolutionary ecology of parthenogenetic strains of the prosobranch, Potamopyrgus antipodarum (Gray) (= P. jenkinsi (Smith)). Malacological Review in press.

Hunter, R. W. 1961. Life cycles of four freshwater snails in limited populations on Loch Lomond, with a discussion of intraspecific variation. Proceedings of the Zoological Society of London 136(4):

Hylleberg, J. 1975. The effect of salinity and temperature on egestion in mud snails (Gastropoda:Hydrobiidae). A study on niche overlap. Oecologia 21:279-289.

Hylleberg, J. 1986. Distribution of hydrobiid snails in relation to salinity, with emphasis on shell size and co-existence of the species. Ophelia Supplement 4: 85-100.

Hylleberg, J .1988. Succession of postglacial mud snails (Hydrobiidae) with notes on morphological types of Hydrobia ulvae and larval shells of three species. Kieler Meeresforsch., Sonderh. 6: 395-403.

Hylleberg, J. and H. R. S. 1987. Niche overlap in mud snails (Hydrobiidae): freezing tolerance. Marine Biology 94: 403-407.

Ibanez, M. and M. R. Alsonso. 1977. Geographical distribution of Potamopyrgus jenkinsi Smith 1889 (Prosobranchia: Hydrobiidae) in Spain. Journal of Conchology 29: 141-146.

Jackiewcz, M. 1973. New stations with Potamopyrgus jenkinsi in Poland and remarks about distribution. Przegl: 364-366.

Jacobsen, R., V. E. Forbes, et al. 1996. Genetic population structure of the prosobranch snail Potamopyrgus antipodarum (Gray) in Denmark using PCR-RAPD fingerprints. Proceedings of the Royal Society of London, Series B in press.

Jacobsen, R., V. E. Forbes, et al. 1997. Clonal variation in life-history traits and feeding rates in the gastropod, Potamopyrgus antipodarium: Performance across a salinity gradient. Functional Ecology.

James, M. R. 1985. Changes in the faunal composition of two thermal streams near Taupo, New Zealand. N. Z. J. Mar. Fresh. Res. 19: 439-443.

James, M. R. 1995. Review of the literature on the effects of water turbidity and sediment on macroinvertebrates in lakes. NIWA, Christchurch. Miscellaneous. Report No. 276.

Jellyman, D. J. 1989. Diet of two species of freshwater eel (Anguilla spp.) in Lake Pounui, New Zealand. New Zealand Journal of Marine and Freshwater Research 23: 1-10.

Johnson, B. R. 1981. The population genetics, taxonomy and ecology of some British and Australian hydrobiid snails with particular reference to the colonizing prosobranch Potamopyrgus jenkinsi (Smith). PhD dissertation, University of York.

http://www.Potamopyrgus/mud snail/exotic species/invasion

Johnson, S. G., Lively, C. M., and S. J. Scrag. 1995. Evolution and ecological correlates of uniparental reproduction in freshwater snails. Experientia. 51(5): 498-509.

Jokela, J. and C. M. Lively. 1995. Spatial variation in infection by digenetic trematodes in a population of freshwater snails (Potamopyrgus antipodarum). Oecologia. 103:509-517.

Jokela, J. and C. M. Lively. 1995. Parasites, sex, and early reproduction in a mixed population of freshwater snails. Evolution 49: 1268-1271.

Jokela, J., Lively, C. M. , Fox, J. A., and M. F. Dybdahl. 1997. Flat reaction norms and frozen phenotypic variation in clonal snails (Potamopyrgus antipodarum). Evolution 51: 1120-1129.

Jokela, J., Lively, C. M. , Fox, J. A., and M. F. Dybdahl. 1997. Evidence for a cost of sex in the freshwater snail Potamopyrgus antipodarum. Ecology 78: 452-460.

Jokela, J., C. M. Lively, M. F. Dybdahl. 1999. Habitat- specific variation in life history traits, clonal population structure and parastism in freshwater snail (Potamopyrgus antipodarums). Journal of Evolutionary Biology 12(2): 350-360.

Jokela, J., C. M. Lively, J. Taskinen, and A. D. Peters. 1999. Effects of starvation on parasite-induced mortality in a freshwater snail (Potamopyrgus antipodarum). Oecologia 119(3): 320-325.

Jokela, J. and S. Negovetic. 2000. Food choice behavior may promote habitat specificity in mixed populations of clonal and sexual Potamopyrgus antipodarum. Animal Behavior 60(4): 434-441.

Jowett, J. G., Richardson, J., Biggs, B. J., Hickey, C. W., and J. M. Quinn. 1991. Microhabitat preferences of benthic invertebrates and the development of generalized Deleatidium spp. habitat suitability curves, applied to four New Zealand rivers. N. Z. J. Mar. Freshwater Res. 25(2): 187-200.

Kabat, A. R. 1993. The prosobranch snail family Hydrobiidae (Gstropoda: Rissooidea): Review of classification and supraspecific taxa. Smithsonian Contributions to Zoology Number 547.

Kennard, A. S. 1941. The geological record of Potamopyrgus jenkinsi (Smith) in the British Isles. Proceedings of the Malacological society of London 24:156.

Kjeldsen, K. 1996. Regulation of algal biomass in a small lowland stream: field experiments on the role of invertebrate grazing, phosphorus, and irradiance. Freshwater Biology 36: 535-546.

Klekowski, R. Z. and A. Duncan. 1966. The oxygen consumption in saline water of young Potamopyrgus jenkinsi (Smith) (Prosobranchiata). Verhandlungen der Internationalen Vereinigung fur theoretishce und angewandte Limnologie 16: 1753-1760.

Krist, A. C. and C. M. Lively. 1998. Experimental exposure of juvenile snails (Potamopyrgus antipodarium) to infection by trematode larvae (Microphallus sp.) infectivity, fecundity compensation, and growth.

Kuris, A. M. and K. D. Lafferty. 1994. Community structure: larval trematodes in snail hosts. Annual Review of Ecology and Systematics 25: 189-217.

Langenstein, S. and P. A. Bowler. 1990. On-going macroinvertebrate analysis using the biotic condition index and the appearance of Potamopyrgus antipodarum (Gray) in Box Canyon Creek, Southern Idaho. Proceeding of the Desert Fishes Council 21: 183-194.

Lassen, H. H. 1975. The diversity of freshwater snails in view of the equilibrium theory of island biogeography. Oecologia 19: 1-8.

Lassen, H. H. 1978. The migration potential of freshwater snails exemplified by the dispersal of Potamopyrgus jenkinsi. Natura Jutlandica 20: 237-241.

Lassen, H. H. 1979. Reproductive effort in Danish mudsnails (Hydrobiidae). Oecologia 40:365-369.

Lassen, H. H. 1979. Electrophoretic enzyme patterns and breeding experiments in Danish mudsnails (Hydrobiidae). Ophelia 18(1): 83-87.

Lassen, H. H. 1978. Tolerance to abiotic factors in mudsnails (Hydrobiidae). Natura Jutlandica 20: 243-250.

Leger, N. and P. Leger. 1974. L'extenstion de Potamopyrgus jenkinsi en Corse. Annales de Parasitologie humaine et comparee 49: 343-347.

Leppaekoski, E. and S. Olenin. 2000. Non-native species and rates of spread: lessons from the brackish Baltic Sea. Biological Invasions 2(2): 151-169.

Levri, E. P. 1996. The effects of size, reproductive condition, and parasitism on foraging behavior in a freshwater snail, Potamopyrgus antipodarum. Animal Behavior 51(4): 891-901.

Levri, E. P 1998. The influences of non-host predators on parasite-induced behavioral changes in a freshwater snail. Oikos 81: 531-537.

Levri, E. P 1998. Perceived predation risk, parasitism, and the foraging behavior of a freshwater snail (Potamopyrgus antipodarum). Canadian Journal of Zoology 76(10): 1878-1884.

Leach, J. H. 1995. Non-indigenoous species in the Great Lakes: were colonization and damage to ecosystem health predictable? J. Aquat. Ecosys. Health. 4(2): 117-128.

Lively, C. M. 1987. Evidence from a New Zealand snail for the maintenance of sex by parasitism. Nature 328(6): 519-521.

Lively, C. M. 1989. Adaptation by a parasitic trematode to local populations of its host. Evolution 46: 1663-1671.

Lively, C. M. 1992. Parthenogenesis in a freshwater snail: reproductive assurance versus parasitic release. Evolution 46: 907-913.

Lively, C. M. 1996. Host-parasite coevolution and sex. BioScience 46: 107-113.

Lively, C. M. and J. C. McKenzie. 1991. Experimental infection of a freshwater snail, Potamopyrgus antipodarum with a digenetic trematode, Microphallus sp. New Zealand Natural Sciences 18(0): 59-62.

Lively, C. M. a. S. G. J. 1994. Brooding and the evolution of parthenogenesis: stategy models and evidence form aquatic invertebrates. Proceedings of the Royal Society of London 256: 89-95.

Lively, C. M. and J. Jokela. 1996. Clinal variation for local adaptation in a host-parasite interaction. Proceedings Royal Soc. London B 263: 8891-897.

Lively, L. E. and P. C. M. 1996. The effects of size, reproductive condition, and parasitism on foraging behavior in a freshwater snail, Potamopyrgus antipodarum. Animal Behavior 51: 891-901.

Locke, A., Reid, D. M., van Leeuwen, H. C., Sprules, W. G., and J.T. Carlton. 1993. Ballast water exchange as a means of controlling dispersal of freshwater organisms by ships. Can. J. Fish. Aquat. Sciences 50: 22086-2093.

Lodge, D. M., K. M. Brown, S. P. Klosiewski, R. A. Stein, A. P. Covich, B. K. Leathers, and C. Bronmark. 1987. Distribution of freshwater snails: Spatial scale and the relative importance of physiochemical and biotic factors. American Malacological Bulletin 5(1): 73-84.

Lumbye, J. 1958. The oxygen consumption of Theodoxus fluviatilis (L.) and Potamopyrgus jenkinsi (Smith) in brackish and fresh water. Hydrobiologia 10: 245-262.

MacArthur, C. P. and D. W. Featherston. 1976. Suppression of egg production in Potamopyrgus antipodarum (Gastropoda: Hydrobiidae) by larval trematodes. New Zealand Journal of Zoology 3: 35-38.

Marsh, P. C. 1980. An occurence of high behavioral drift for a stream gastropod. The American Midland Naturalist 104(2): 410-411.

Martinez-Lopez, F., J. Jimenez, J. Subias, and J. F. Amela. 1986. Sobre la distribucion de Potamopyrgus jenkinsi (Smith, 1889) (Gastropoda: Prosobranchia) en Las Cuencas De Los Rios Mijares, Turia Y Jucar. Iberus 6(2): 245-255.

McCarter, N. H. 1986. Food and energy in the diet of brown and rainbow trout from Lake Benmore, New Zealand. N. Z. J. Mar. Fresh. Res. 20: 551-559.

McDowall, R. 1990. New Zealand freshwater fishes: a natural history and guide. Publ. Reed, Auckland, New Zealand.

McFarlane, W. V. 1945. The life cycle of the heterophyoid trematode Telegaster opistorchis. Trans. R. Soc. N. Z. 75: 218-230.

Meenakshi, V. R., P. E. Hare, et al. 1969. The chemical composition of the periostracum of the molluscan shell. Comparative Biochemistry and Physiology 29: 611-620.

Michaut, P. 1968. Donne biologiques sur un Gasteropode Prosobranche recmment introduit en Cote-d'Or, Potamopyrgus jenkinsi. Hydrobiologia 32: 513-527.

Mills, E. L., Leach, J. H., Carlton, J. T., and C. L. Secor. 1993. Exotic species in the Great Lakes: a history of biotic crises and anthropogenic introduction. J. Great Lakes Res. 19(1): 1-54.

Modena, P. and P. Turin. 1991. Two new locations for Potamopyrgus jenkinsi (Smith), new record in Italy. Bolletino Malacologico. 27(5-9): 141-143.

Moller, V., V. E. Forbes, and M. H. Depledge. 1994. Influence of acclimation and exposure temperature on the accute toxicity off cadmium to the freshwater snail Potamopyrgus antipodarium (Hydrobiidae). Environmental Toxicity and Chemistry 13(9): 1519-1524.

Murphey, K.A., M.J. Freeman, and P. Bowler. 1993. Valley of the Mighty Snake: An overview of the Cultural and Natural history of Hagerman Valley, Southwestern Idaho. Hagerman Valley Historical Society Report No. 1. J & D Printers, Castleford, Idaho. (Book)

Muss, B. J. 1967. The fauna of Danish estuaries and lagoons. Medd. Danm. Fishk. Havunders, N.S. 5: 1-316.

Muzika, R. M. and A. M. Liebhold. 1997. Ecological effects of introduced species in North America. Acta Phytopatholoica et Entomologica. 32 (1-2)161-170.

Okland, J. 1979. Distribution of environmental factors and freshwater snails (Gastropoda) in Norway: use of European invertebrate survey principles. Malacologia. 18: 211-222.

Okland, J. 1983. Factors regulating the distribution of fresh-water snails (Gastropoda) in Norway. Malacologia 24: 277-288. Obdrlik, P. and U. Fuchs.

Okland, J. 1991. Surface water connection and the macrozoobenthos of two types of floodplains on the Upper Rhine. Regulated Rivers Research and Management.6(4): 279-288.

Oregon Sea Grant . 2000. Oregon (aquatic nuisance species). Aquatic Nuisance Species Report: an update on Sea Grant research and outreach projects, pp. 209-212.

Parker, E. D. and M. Niklasson. 1996. Genetic structure and evolution in parthenogenetic animals. Evolutionary Genetics from Molecules to Morphology (eds. R. Singh and C. Krimbas) in press, Cambridge University Press, Cambridge.

Patil, A. M. 1958. The occurance of a male of the prosobranch Potamopyrgus jenkinsi (Smith) var. carinata (Marshall) in the Thames at Sonning, Berkshire. Ann. and Mag. Nat. Hist. 13: 232-240.

Phillips, N. R. and D. M. Lambert. 1989. Genetics of Potamopyrgus antipodarum (Gastropoda:

Prosobranchia): Evidence for reproductive modes. New Zealand Journal of Zoology 16: 435-445.

Polishchuk, V. V. and Ljurin. 1976. On finding Potamopyrgus jenkinsi (Smith 1889) (Gastropoda: Mollusca) in the Pripjat Polessie. Dopovidi Akademiyi nauk Ukrayins'koyi RSR (ser B) 4: 364-366.

Ponder, W. F. 1988. Potamopyrgus antipodarum a Molluscan colonizer of Europe and Australia. J. Molluscan Studies 54: 271-286.

Quinn, J. M., Steele, G. L., Hickey, C. W., and M. L. Vickers. 1994. Upper thermal tolerance of twelve New Zealand stream invertebrate species. N. Z. J. Mar. Fresh. Res. 28(4): 391-397.

Quinn, G. P. Lake, P. S., and E. S. G. Schreiber. 1996. Littoral benthos of a Victorian lake and its outlet stream: spatial and temporal variation. Australian J. Ecology 21: 292-301.

Real, G. 1973. Reparition en France de Potamopyrgus jenkinsi (E.A. Smith, 1889). Haliotis 3: 199-204.

Reavell, P. E. 1980. A study on the diets of some British freshwater gastropods. J. Conchology. 30: 253-271.

Rhein, A. (1937). Diploide parthenogenesis bei Hydrobia jenkinsi (Smith) (Prosobranchia). Naturwissenschaften 23: 100.

Ribi, G. 1982. Benthic faunas of forested streams and suggestions for their management. N. Z. J. Ecology. 5: 140-150.

Ribi, G .1983. The formation, structure and utilization of stone surface organic layers in two new Zealand streams. Freshwater Biology 13: 57-72.

Ribi, G. 1986. Within-lake dispersal of the prosobranch snails, Viviparus ater and Potamopyrgus jenkinsi. Oecologia 69: 60-63.

Ribi, G. and H. Arter. 1986. Sex related difference of movement speed in the freshwater snail, Viviparus ater. Journal of Molluscan studies.

Richards, D. C., Cazier, L.D., and G. T. Lester. 2001. Spatial distribution of three snail species including the invader Potamopyrgus antipodarum in a freshwater spring. Western North American Naturalist. 61(3): 375-380.

Roth, G. 1987. Contribution to the distribution and biology of Potamopyrgus jenkinsi E.A. Smith, 1889, in the Rhine River catchment areas (Prosobranchia: Hydrobiidae). Arch. Hydrobiol. Suppl. 79: 49-68.

Rounick, J. S. and M. J. Winterbourn. 1982. Benthic faunas of forested streams and suggestions for their management. New Zealand Journal of Ecology 5: 140-150.

Ryan, P. A. 1982. Energy contents of some New Zealand freshwater annimals. New Zealand Journal of Marine and Freshwater Research 16: 283-287.

Sanderson, A. R. 1940. Maturation in the parthenogenetic snail Potamopyrgus jenkinsi (Smith) and the snail Peringia ulvae (Pennant). Proceedings of the Zoological Society of London, Series A 110: 11-15.

Savage, A. A. 1996. Density dependent and density independent relationships during a twenty-seven year study of the population dynamics of the benthic macroinvertebrate community of a chemically unstable lake. Hydrobiologia 335: 115-131.

Scarsbrook, M. R. and C. R. Townsend. 1993. Stream community structure in relation to spatial and temporal variation: a habitat template study of two contrasting New Zealand streams. Freshwater Biology. 29: 395-410.

Schreiber, E. S. G., A. Glaister, G. P. Quinn, and P. S. Lake. 1998. Life history and population dynamics of the exotic snail Potamopyrgus antipodarum (Prosobranchia: Hydrobiidae) in Lake Purrumbete, Victoria, Australia. Marine and Freshwater Research 49(1): 73-78.

Scott, D., J. W. White, et al. 1994. Invertebrate fauna of three streams in relation to land use in Southland, New Zealand. New Zealand Journal of Marine and Freshwater Research 28: 277-290.

Scott, R. R. and Tomkins, A. R.1986. Effects of treated sewage effluent on the macroinvertebrates of a fine sediment substrate stream. Mauri Ora 13: 1-12.

Scrimgeour, G. J. 1989. Effects of bleached kraft mill efluent on macroinvertebrate and fish populations in weedbeds in a New Zealand hydro-electric lake. New Zealand Journal of Marine and Freshwater Research 23: 373-379.

Scrimgeour, G. J. and M. J. Winterbourn. 1989. Effects of floods on epilithon and benthic macroinvertebrate populations in an unstable New Zealand river. Hydrobiologia. 171: 33-44.

Sheldon, F. W. 1997. Changes in biofilms induced by flow regulation could explain extinctions of aquatic snails in the lower River Murray, Australia. Hydrobiologia 347: 97-108.

Siegismund, H. R. a. J. H. 1987. Dispersal-mediated coexistence of mud snails (Hydrobiidae) in an estuary. Marine Biology 94: 395-402.

Simpson, J. F. 1976. On the existence of discrete morphological types within the species Potomopyrgus jenkinsi (Smith). Journal of Molluscan Studies 42: 108-113.

Smith, E. A. (1889). Notes on British Hydrobiidae with a description of a supposed new species. Journal of Conchology 6: 142-145.

Smith, E. A. 1907. Notes on Paludestrina jenkinsi. Proc. Malac. Soc. London 7: 203-204.

Smith, E. A. (1889). Notes on British Hydrobiae with a description of a supposed new species. Journal of Conchology 6: 142-145.

Smith, H., Van der Velden, J. A., and A. Klink .1994. Macrozoobenthic assemblages in littoral sediments in the enclosed Rhine-Meuse Delta. Netherland J. Aquatic Ecology. 28(2): 199-212.

Stewart, T. W., J. M. Haynes. 1994. Benthic macroinvertebrate communities of southwestern Lake Ontario following invasion of Dreissena. Journal of Great Lakes Research 20(2): 479-493.

Stojaspal, F. 1975. Potamopyrgus jenkinsi (E.A. Smith 1889) in Oesterreich. Mitt. Deut Malako-zool Ges 3: 243.

Strayer, D. L. 1997. Effects of alien species on freshwater mollusks in North America. Journal of the North American Benthological Society 18(1): 74-98.

Strzelec, M. and M. Krodkiewska. 1994. The rapid expansion of Potamopyrgus jenkinsi (Smith) in Upper Silesia (Southern Poland)(Gastropoda: Prosobranchia: Hydrobiidae). Malkolgische Abhandlungen (Dresden). 17(1-9): 83-86.

Strzelec, M. and W. Serafinski. 1996. Population ecology of Potamopyrgus antipodarum (Gray) in a recently colonized area: Upper Silesia (Southern Poland) (Gastropoda: Prosobranchia: Hydrobiidae). Malakologische Abhandlungen Staatliches Museum fur Tierkunde Dresden 18: 75-82.

Tomkins, A. R. and R. R. Scott. 1986. Effects of treated sewage effluent on the macroinvertebrates of a fine sediment substrate stream. Mauri Ora 13: 1-12.

Talbot, J. M. and J. C. Ward. 1987. Macroinvertebrates associated with aquatic macrophytes in Lake Alexandrina, New Zealand. New Zealand Journal of Marine and Freshwater Research 21: 199-213.

Todd, M. E. 1964. Osmotic balance in Hydrobia ulvae and Potamopyrgus jenkinsi (Gastropoda: Hydrobiidae). Journal of Experimental Biology 41: 665-677.

Towns, D. R. 1979. Composition and zonation of benthic invertebrate communities in a New Zealand kauri forest stream. Freshwater Biology 9: 251-262.

Towns, D. R. 1981. Life histories of benthic invertebrates in a Kauri forest stream in Northern New Zealand. Australian Journal of Marine and Freshwater Research 32: 191-211.

Towns, D. R. 1981. Effects of artifical shading on periphyton and invertebrates in a New Zealand stream. New Zealand Journal of Marine and Freshwater Research 15: 185-192.

van den Berg, M. S., H. Coops, R. Noordhuis, J. van Schie, and J. Simons. 1997. Macroinvertebrate communities in relation to submerged vegetation in two chara-dominated lakes. Hydrobiologia 342/343: 143-150.

Vareille-Morel, C. 1983. Les mouvements journaliers du mollusque Potamopyrgus jenkinsi Smith. Etude sur le terrain et en laboratoire. Haliotis 13: 31-34.

Vareille-Morel, C. 1983. Les mouvements journaliers du mollusque Potamopyrgus jenkinsi Smith. Etude sur le terrain et en laboratoire. Haliotis 13: 31-34.

Wallace, C. 1969. Water temperature as a factor limiting the distribution of Potamopyrgus antipodarum (Gastropoda: Prosobranchia) in the New Zealand thermal region. N. Z. J. Mar. Freshwater Res. 3: 453-458.

Wallace, C. 1970. The New Zealand species of Potamopyrgus (Gastropoda: Hydrobiidae). Malacologia 10: 283-321.

Wallace, C. 1970. Population studies on the New Zealand freshwater gatropod Potamopyrgus antipodarum (Gray). Proc. Malacol. Soc. Lond. 39: 139-149.

Wallace, C. 1972. Morphological variation of Potamopyrgus jenkinsi (Smith) from England and a comparison with the New Zealand species Potamopyrgus antipodarum (Gray). Proc. Malacological Soc. London 40: 133-145.

Wallace, C. 1973. A guide to the freshwater Molluska of New Zealand. Tuatara. 20:141-159.

Wallace, C. 1974. Larval Trematoda parasitizing the New Zealnand species of Potamopyrgus (Gastropoda: Hydrobiidae). Mauri Ora. 2: 17-30.

Wallace, C. 1978. The macroinvertebrate fauna of a New Zealand forest stream. N. Z. J. Zoology 5: 157-169.

Wallace, C. 1978. Notes on the distribution of sex and shell characters in some Australian populations of Potamopyrgus (Gastropoda: Hydrobiidae). Journal of the Mallacological Society of Australia 4((1-2)): 71-76.

Wallace, C. 1979. Notes on the occurrence of males in populations of Potamopyrgus jenkinsi. Journal of Molluscan Studies 45: 61-67.

Wallace, C. 1985. On the distribution of sexes of Potamopyrgus jenkinsi (Smith). Journal of Molluscan studies 51: 93-107.

Wallace, C. 1992. Parthenogenesis, sex, and chromosomes in Potamopyrgus. Journal of Molluscan Studies 58: 93-107.

Walter, J. E. 1980. Eine neue Schneckenart im Zurichsee: Potamopyrgus jenkinsi (Gastropoda: Prosobranchia). Vjschr Naturforsch Ges Zurich 125: 197-200.

Warwick, T. 1944. Inheritance of the keel in Potamopyrgus jenkinsi (Smith). Nature 154: 798-799.

Warwick, T. 1969. Systematics of the genus Potamopyrgus (Hydrobiidae) in Europe, and the causation of the keel in this snail. Malacologia 9: 301-302.

Warwick, T. 1969. Strains in the mollusc Potamopyrgus jenkinsi (Smith). Nature 169: 551-552.

Watton, A. J. and H. A. Hawkes. 1984. Studies on the effects of sewage effluent on gastropod populations in experimental streams. Water Research 18: 1235-1248.

Winterbourn, M. J. 1969. Water temperature as a factor limiting the distribution of Potamopyrgus antipodarum (Gastropoda-Prosobranchia) in the New Zealand thermal region. New Zealand Journal of marine and freshwater research 3: 453-458.

Winterbourn, M. J. 1970. The New Zealand species of Potamopyrgus (Gastropoda: Hydrobiidae). Malacologia 10(2): 283-321.

Winterbourn, M. J. 1970. Population studies on the New Zealand freshwater gastropod, Potamopyrgus antipodarum (Gray). Proceedings of the Malacological Society of London 39: 139-149.

Winterbourn, M. J. 1972. Morphological variation of Potamopyrgus jenkinsi (Smith) from England and a comparison with the New Zealand species, Potamopyrgus antipodarum (Gray). Proceedings of the Malacological Society of London 40: 133-145.

Winterbourn, M. J. 1973. A guide to the freshwater mollusca of New Zealand. Tuatara 20:141-159.

Winterbourn, M. J. 1973. Larval Trematoda parasitizing the New Zealand species of Potamopyrgus (Gastropoda: Hydrobiidae). Mauri Ora 2: 17-30.

Winterbourn, M. J. 1978. The macroinvertebrate fauna of a New Zealand forest stream. New Zealand Journal of Zoology 5: 157-169.

Winterbourn, M. J. and M. H. Lewis (?). Littoral fauna. New Zealand Lakes, edited by V. H.. Jolly and J. M. A.. Brown: 271-281.

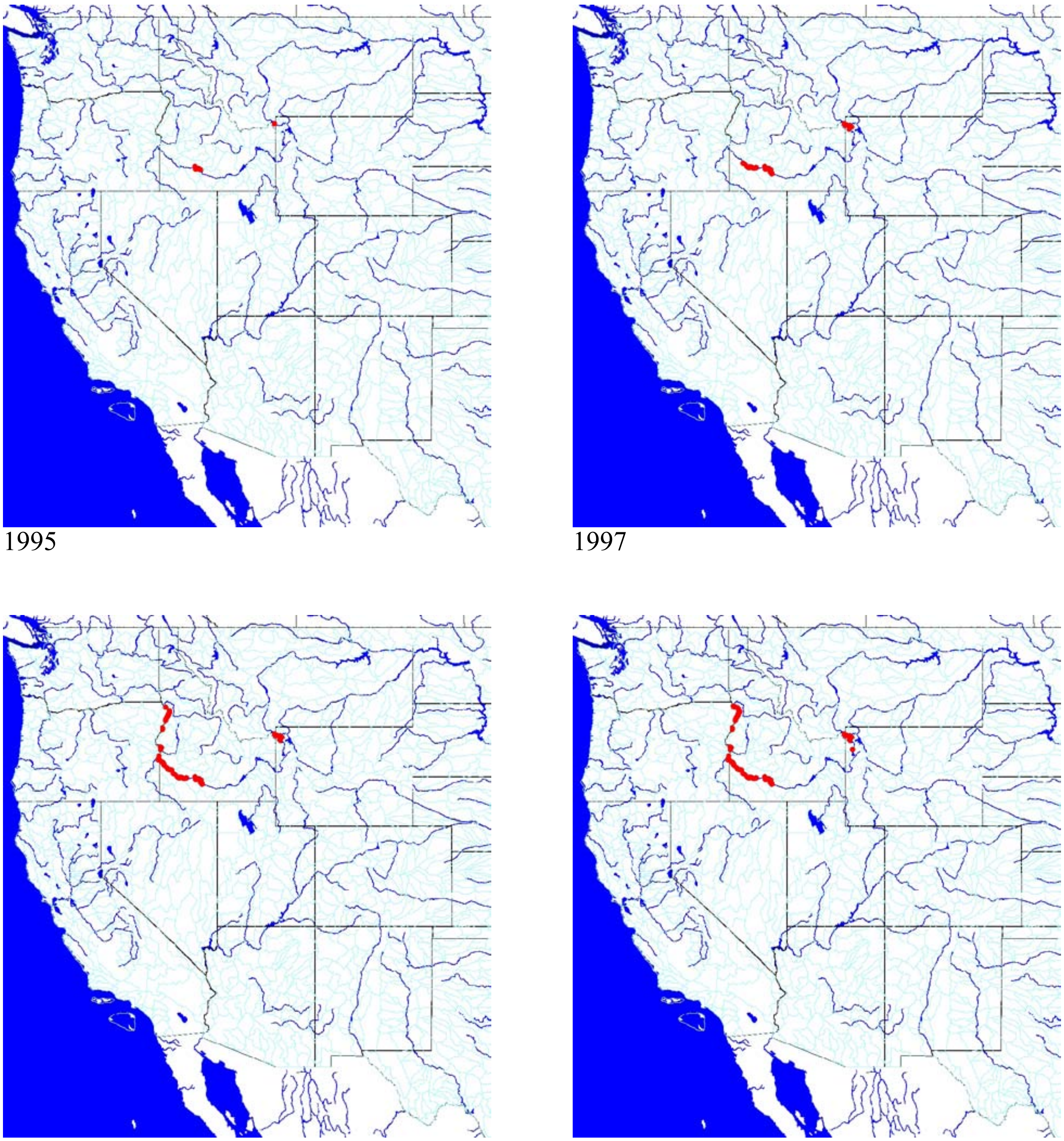
Winterbourn, M. J., Rounick, J. S. and B. Cowie. 1981. Are New Zealand stream ecosystems really different? N. Z. J. Mar. Fresh. Res. 15: 321-328.

Winterbourn, M. J. and A. F. 1989. Effects of nutrient enrichment and grazing on periphyton assemblages in some spring-fed, South Island streams. New Zealand Natural Sciences 16: 57-65.

Winterbourn, M. J. and P. A. Ryan. 1994. Mountain streams in Westland, New Zealand: Benthic ecology and management issues. Freshwater Biology. 32: 359-373.

Zaranko, D. T., D. G. Farara, F. G. Thompson (1997). Another exotic mollusc in the Laurentian Great Lakes: the New Zealand native Potamopyrgus antipodarum (Gray 1843) (Gastropoda, Hydrobiidae). Canadian Journal of Fisheries and Aquatic Sciences 54: 809-814.

Figure 4. Distribution map – Progression and current distribution (December 2003) of the New Zealand mudsnail, *Potamopyrgus antipodarum*, in the western U.S. (west of the 100th meridian).



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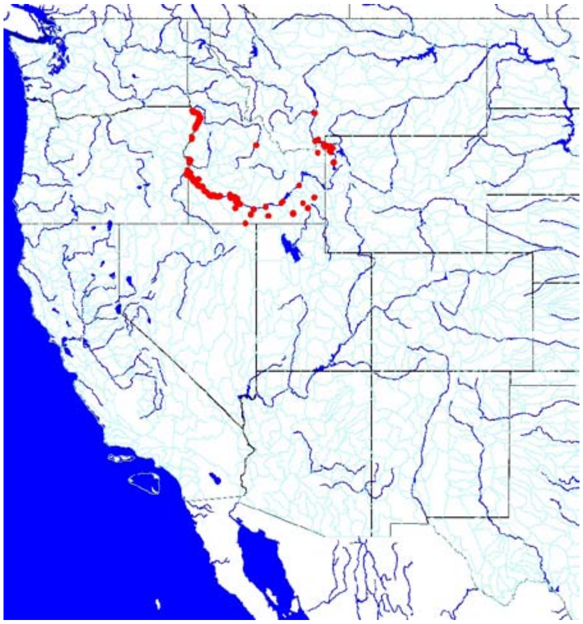

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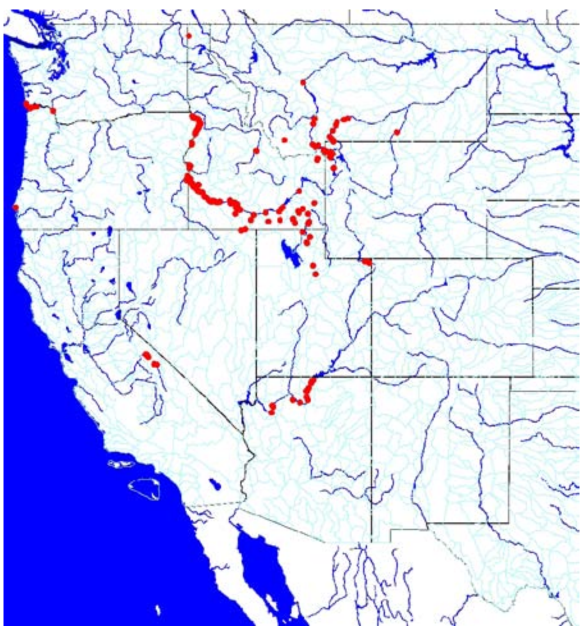

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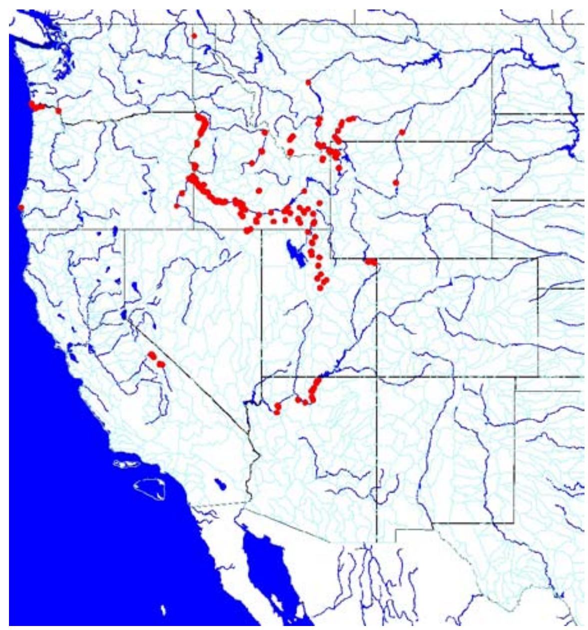

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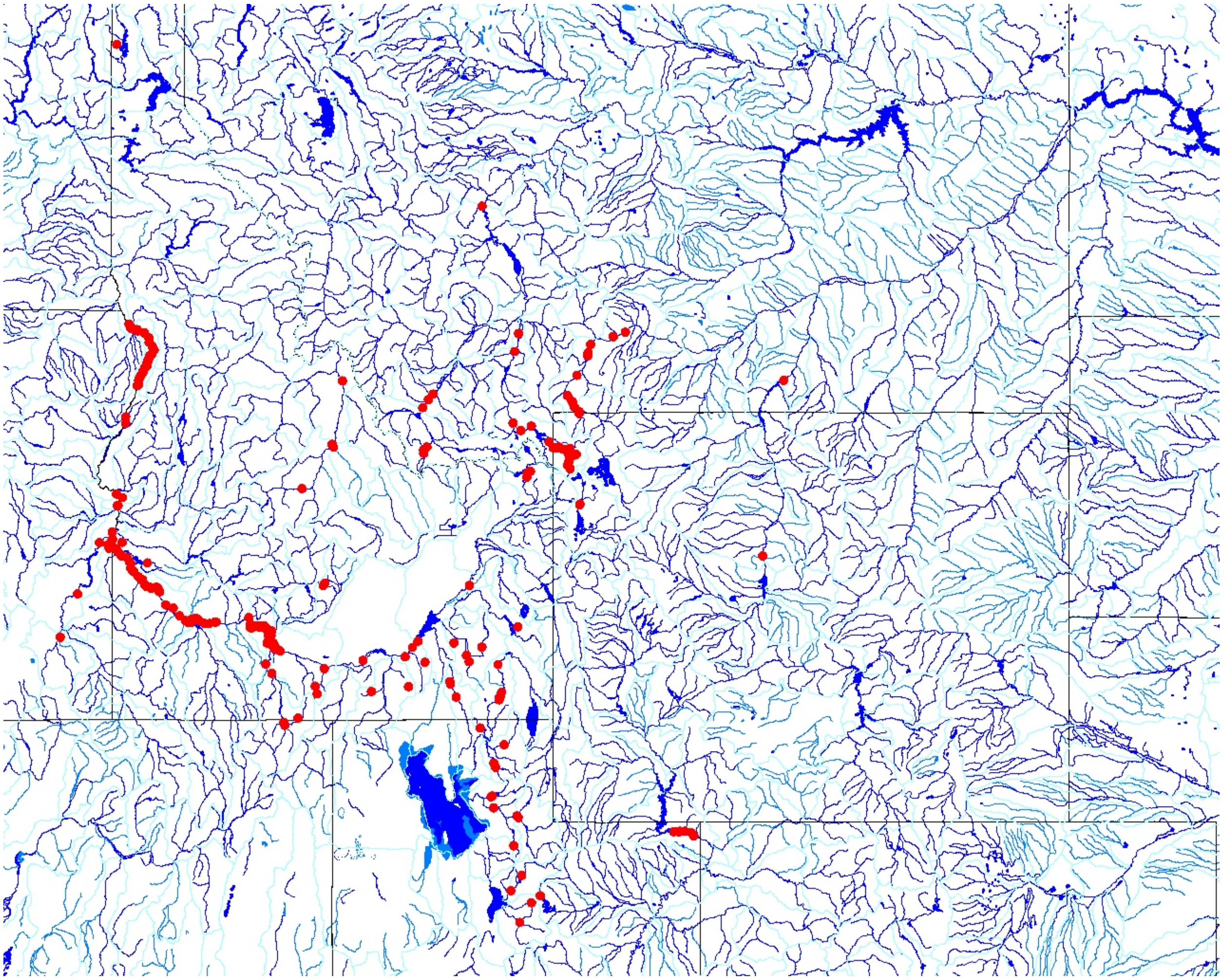
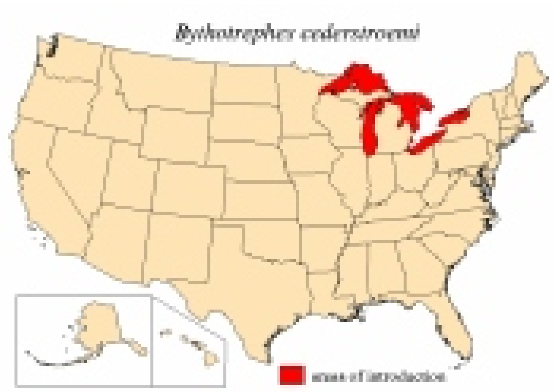


Figure 19. Distribution of New Zealand mudsnail in Idaho, Utah, Wyoming, and Montana relative to western reaches of the Missouri River headwaters.

[5]***Bythotrephes cederstroemi*** (Schodler, 1877)

Common name: Spiny water flea

Introduction: The native range of the spiny water flea, *Bythotrephes cederstroemi*, is the northern and central Palearctic (northern Europe to Caspian Sea). Although ballast water may have been the source of introduction, it is unclear whether this presumptive source was the root



cause in the pioneering event, since most ocean-going ships do not carry freshwater in ballast. In North America, the spiny water flea was first collected in 1984 from Lake Huron (Bur et al.1986), followed in 1985 when the species was collected from Lake Erie and Lake Ontario. In 1986 the spiny water flea was collected from Lake Michigan, and in 1987 the species had successfully invaded Lake Superior. More recently it has been found in several small lakes in Minnesota and in more than a dozen lakes in Ontario, Canada. Researchers suspect *Bythotrephes cederstroemi* could be responsible for the decline of 3 daphnia species in Lake Michigan (Lehman 1991).

Other nonindigenous crustaceans in the United States. Besides *Bythotrephes cederstroemi*, there are numerous crusteceans that have gained increasing attention with respect to their being invasives and potentially having adverse impacts on native fauna. Here, we consider only a few species that might be considered in subsequent investigations of species invasions of the Red River basin. Throughout the US, there are no fewer than 77 species of crustaceans that are considered nonindigenous to the waters in which they occur, and of that number, 44 have become established in their new environment. Crustaceans are found in every kind of aquatic

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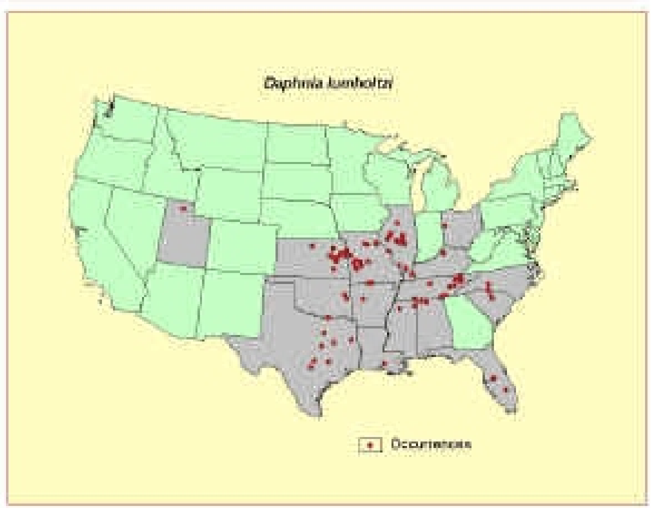
[5]Original material authored by Amy Benson and Pam Fuller, USGS/BRD/FISC. Updated, expanded, and edited January, 2004.

habitat, and marine and estuarine habitats outnumber freshwater areas (lentic or lotic) habitats as far as species incursions, over two dozen introduced species are found in fresh water. Regardless of the receiving habitats, these introductions are linked to worldwide sources. Introductions of crustaceans as early as 1873, have typically occurred through aquaculture and research escapes and releases, ballast water discharge, ship fouling, stocking for food or gamefish forage, and stock contamination with fish or oyster species. Studies have shown that nonindigenous crustaceans impact food webs and fish communities, exclude native congeners, and alter habitats.

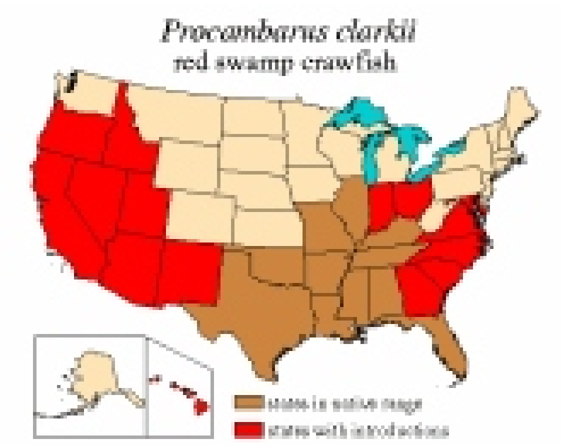
***Daphnia lumholtzi* Sars.** The native range of *Daphnia lumholtzi*are freshwater lakes of east Africa, southwest Asia, and east Australia (Havel 1993). It was first reported in Stockton Lake in southwest Missouri in 1991 and can be found in 13 states from Illinois and Ohio down to Texas



across to North Carolina and south to Florida. *D. lumholtzi* has spread much faster than another zooplankton invader, *Bythotrephes cederstroemi,*for reasons unknown. The method of introduction is uncertain. But, coincidentally, one of its first appearances was a lake in Texas where the Nile perch was first introduced to North America (Havel 1993). It is well established in the U.S. Effects or impacts are yet unknown (see USGS/BRD/FISC posting last accessed December 8, 2004 at http://nas.er.usgs.gov/crustaceans/docs/dlumholtzi.html).

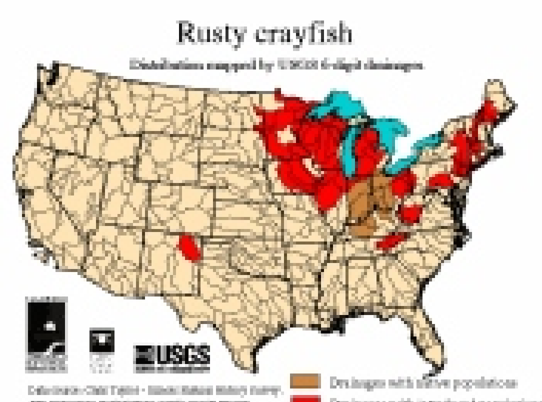
Procambarus clarkii (Red Swamp Crawfish). The red swamp crawfish is native to fresh waters from northern Mexico to the Florida panhandle and north to southern Illinois and Ohio (Hobbs 1989). This crayfish has been accidentally and deliberately introduced well outside its range in the Americas and in Africa and Asia (Fitzpatrick 1983). In the United States, it has been introduced into at least 15 states (Hobbs 1989). This species and one other, the white river crawfish (*P.*

acutus), are the cultured crawfish in "Cajun" crawfish dishes. The species are very similar and the two species comprise over 90% of the crawfish produced in the U.S. The way to distinguish the two species is the presence of a blue vein under the tail of the red swamp crawfish; it is absent in the white river crawfish. In California, wild populations of the red swamp crawfish eat rice crops (Pennak 1989). They have also been found to prey on California newts (*Taricha torosa*) and may



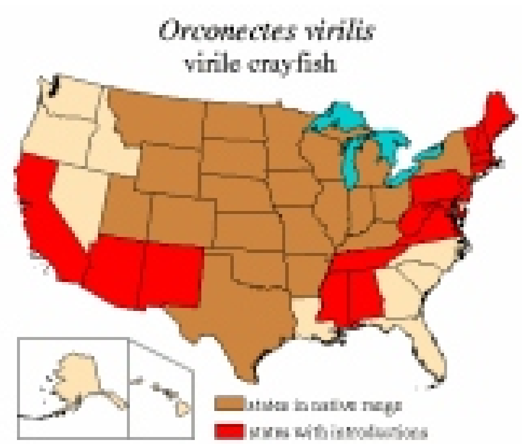
be responsible for their decline in some areas (Gamradt and Kats 1996).

Orconectes rusticus (Rusty Crayfish). The rusty crayfish is native to western Ohio, eastern Indiana, and Kentucky. It has become established in 17 states outside of its native range, primarily through bait release by anglers. Contrary to some reports, it is not present in Missouri or central



Tennessee (C. Taylor, personal communication). Rusty crayfish often displace native crayfish including *O. propinquus, O. virilis*, and *O. sanborni*, reduce the amount and kinds of aquatic plants and invertebrates, and reduce some fish populations (Gunderson 1995). In Wisconsin, where the rusty crayfish has been introduced, it is hybridizing with the blue crayfish (*O. propinquus*), another introduced species. The resulting hybrids are outcompeting both parental species for both food and shelter (Roush 1997). It has been estimated that rusty crayfish may consume twice as much food as the native virile crayfish, *O. virilis* (Momot 1992). This crayfish may affect fish populations by competing with juvenile fish for food and by preying on fish eggs (Gunderson 1995).

Orconectes virilis (Virile Crayfish). The virile crayfish is native from Saskatchewan to Ontario, Canada, and from Montana and Utah to Arkansas, New York, and possibly southwestern Maine. It has been introduced into 17 states outside its native range, largely through bait-bucket releases which are frequent modes of "jump" dispersal events for crusteceans and other aquatic biota. This species is also commonly sold by biological supply houses and could be dispersed this way as



well. In California, the virile crayfish may be responsible for the decline of native Shasta crayfish (*Pacifastacus fortis*). *O. virilis*was introduced into California in the early 1960s from east of the Continental Divide. It has a higher fecundity and more rapid growth rate than the native Shasta crayfish. The virile crayfish matures at one year and produces up to 443 eggs while the Shasta crayfish matures at 4 years and produces 10-70 eggs. As a result, the virile crayfish is replacing the native species (Light et al. 1995). Introductions of this species have also displaced native crayfish species in the Patapsco River, Maryland (Schwartz et al. 1963).

Representative biota of concern: Fishes

[6]***Dorosoma cepedianum* (Lesueur 1818)**

Common Names: gizzard shad (hickory shad)

Size: 52 cm.

Taxonomy and identification. Various sources document the taxonomy of the gizzard shad, including Becker (1983); Whitehead (1985); Page and Burr (1991); Etnier and Starnes (1993).

Native Range. Figure 5 depicts the presumptive original distribution for the gizzard shad, although the historic distribution is unclear for certain localities, i.e. St. Lawrence-Great Lakes. The species remains widespread in the Mississippi, Atlantic, and Gulf Slope drainages from Quebec to central North Dakota and New Mexico, and south to central Florida and Mexico (Page

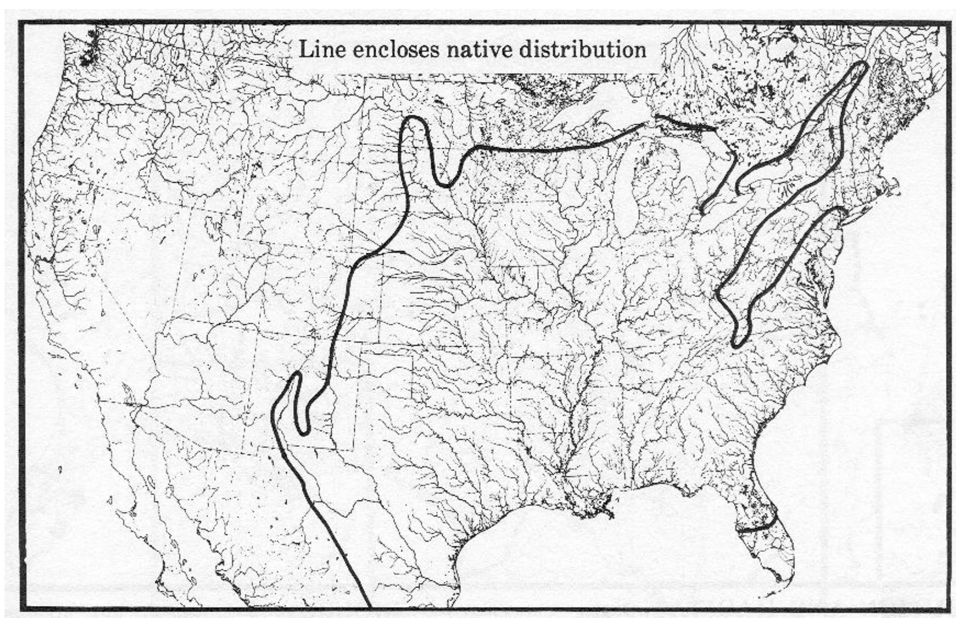


Figure 5. Presumptive native distribution of gizzard shad (after Lee et al 1980).

[6]Original material prepared by Pam Fuller (January 13, 2000) at the Center for Aquatic Resource Studies, USGS/BRD/FISC. The Center is part of the Biological Resources Division of the Geological Survey within the U.S. Department of the Interior. Updated and edited January, 2004.

and Burr 1991). Propst and Carlson (1986) believe the gizzard shad may be native to the South Platte drainage in Colorado. Since the 1600's, gizzard shad has expanded its range naturally to include Massachusetts (O'Leary and Smith 1987; Hartel 1992), and the species has been introduced to Lake Erie upon completion of the Ohio Canal (Jordan 1882). Cold weather apparently limits the species' northern range (Becker 1983).

Nonindigenous Occurrences. Figure 6 summarizes the presumptive native distribution overlain with the species' expanded range accountable as introduction. Gizzard shad were introduced into Colorado in the Arkansas, South Platte, and Republican drainages (Beckman 1952; Woodling

Figure 6. Native distribution overlain with expanded introduced range.

1985), although the species may have occurred in the South Platte drainage prior to the documented introduction. In Kansas, gizzard shad have been stocked in Fall River Reservoir, in sand pits along Prairie Dog Creek, and in Kirwin, Webster, and Cedar Bluff reservoirs (Cross 1967), and in Nebraska, one or more unspecified areas (presumably the Platte River) have also been stocked (Bouc 1987). The species has expanded its range into Wyoming from introductions into Nebraska (Baxter and Simon 1970), and from intentional stockings in the state east of the Continental Divide (Hubert 1994). Gizzard shad have become established in the upper Rio Grande River in New Mexico (J. Wilbur, personal communication), and the species has been stocked in Utah (B. Schmidt, personal communication).

East of the Mississippi River, the species, although native to Illinois, has expanded its range and abundance within the state due to construction of reservoirs (Smith 1979). Gizzard shad were stocked in Martin's Fork Lake and in other small impoundments above Cumberland Falls in Kentucky (Minckley and Krumholz 1960; Burr and Warren 1986). It is unclear whether gizzard shad are native to the Great Lakes or gained access through canals and rivers in New York such as the Mohawk River, Oswego River, and Barge Canal (Smith 1985). The species has reached Lake Michigan (Wisconsin) either through the Chicago River Canal or the Fox-Wisconsin Canal at Portage (Becker 1983). Miller (1957) reviewed the evidence for native versus introduced status. As noted previously, gizzard shad gained access to Lake Erie, Ohio via the Ohio Canal (Jordan 1882). Gizzard shad were accidentally stocked in Conowingo Pond, Lancaster County, Pennsylvania (Denoncourt et al. 1975a). They have expanded into Lake Champlain and the Connecticut River, Vermont (K. Cox, personal communication). They were stocked in Lake Anna, Smith Mountain Lake, and Kerr, Philpot, and Leesville reservoirs, Virginia (Jenkins and Burkhead 1994).

Means of Introduction. These fish were stocked intentionally for forage. The Wyoming populations also spread from introductions into Nebraska (Baxter and Simon 1970). In Pennsylvania, gizzard shad were stocked accidentally with American shad (Denoncourt et al. 1975a). In Vermont, gizzard shad have expanded their range through the Connecticut River assisted by fishways that were constructed for American shad *Alosa sapidissima* and Atlantic salmon *Salmo salar* restoration (Cox, personal communication). The species has likely gained access to Lake Champlain through the Hudson Barge Canal that links the lake to the Hudson River (Cox, personal communication), and they have gained access to Lake Michigan through either the Chicago River Canal or the Fox-Wisconsin Canal (Becker 1983), and to Lake Erie through the Ohio Canal (Jordan 1882).

Status: Established in many states.

Impact of Introduction. Competition for food between gizzard shad and other fish species may occur (Burns 1966b; Moyle 1976a). Jenkins (1954) found that gizzard shad directly compete with centrarchids resulting in decreased growth and size of the centrarchids. Gizzard shad show tremendous invasion potential. After only two plantings totally 1020 fish in Lake Havasu, the species spread through the Colorado River from Davis Dam southward to the Mexican border, the Salton Sea, and associated irrigation ditches within only 18 months (Burns 1966b).

References

Baxter, G. T., and J. R. Simon. 1970. Wyoming fishes. Wyoming Game and Fish Department Bulletin 4, Cheyenne, WY. 168 pp.

Beckman, W. C. 1952. Guide to the fishes of Colorado. University of Colorado Museum, Boulder, CO.

Bouc, K. 1987. The fish book. Nebraskaland Magazine 65(1):1-130.

Burr, B. M., and M. L. Warren, Jr. 1986. A distributional atlas of Kentucky fishes. Kentucky Nature Preserves Commission Scientific and Technical Series 4. 398 pp.

Cox, K. nd. Vermont Department of Fish and Wildlife, North Springfield, VT.

Cross, F. B. 1967. Handbook of Fishes of Kansas. State Biological Survey and University of Kansas Museum of Natural History, Miscellaneous Publication 45, Topeka, KS.

Denoncourt, R. F., T. B. Robbins, and R. Hesser. 1975a. Recent introductions and reintroductions to the Pennsylvania fish fauna of the Susquehanna River drainage above Conowingo Dam. Proceedings of the Pennsylvania Academy of Science 49:57-58.

Hartel, K. E. 1992. Non-native fishes known from Massachusetts freshwaters. Occasional Reports of the Museum of Comparative Zoology, Harvard University, Fish Department, Cambridge, MA. 2. September. pp. 1-9.

Hubert, W. 1994. Exotic fish. Pages 158-174 in T. L. Parrish, and S. H. Anderson, editors. Exotic species manual. Wyoming Game and Fish Department, Laramie, WY.

Jenkins, R. E., and N. M. Burkhead. 1994. Freshwater fishes of Virginia. American Fisheries Society, Bethesda, MD.

Miller, R. R. 1957. Origin and dispersal of the alewife, Alosa pseudoharengus, and the gizzard shad, Dorosoma cepecianum, in the Great Lakes. Transactions of the American Fisheries Society 86:97-111.

Minckley, W. L., and L. A. Krumholz. 1960. Natural hybridization between the clupeid genera

Dorosoma and Signalosa, with a report on the distribution of S. petenensis. Zoologica 44(4):171-180.

O'Leary, J., and D. G. Smith. 1987. Occurrence of the first migration of the gizzard shad, (Dorosoma cepedianum), in the Connecticut River, Massachussetts. U.S. National Marine Service Fishery Bulletin 85(2):380-383.

Page, L. M., and B. M. Burr. 1991. A field guide to freshwater fishes of North America north of Mexico. The Peterson Field Guide Series, volume 42. Houghton Mifflin Company, Boston, MA.

Schmidt, B. - Chief Fisheries Mangement, Division of Wildlife Resources, Salt Lake City, UT. Response to NBS-G non-indigenous questionaire. 1992.

Smith, C. L. 1985. The inland fishes of New York state. New York State Department of Environmental Conservation, Albany, NY. 522 pp.

Smith, P. W. 1979. The fishes of Illinois. University of Illinois Press, Urbana, IL.
Wibur, J. - Bureau of Reclamation, Albuquerque, NM.

Woodling, J. 1985. Colorado's little fish: a guide to the minnows and other lesser known fishes in the state of Colorado. Colorado Division of Wildlife, Denver, CO. 77 pp.

[7]***Gila atraria* (Girard 1856)**

Common Name: Utah chub

Taxonomy and identification: Simpson and Wallace (1978); Sigler and Sigler (1987); Page and Burr (1991); Bond (1994).

Size: 56 cm (maximum length)

Native Range. Figure 7 indicates the native range of the Utah chub which includes the upper Snake River system in Wyoming and Idaho, and Lake Bonneville basin (including Great Salt Lake drainage and Sevier River system) in southeastern Idaho and Utah (Page and Burr 1991).

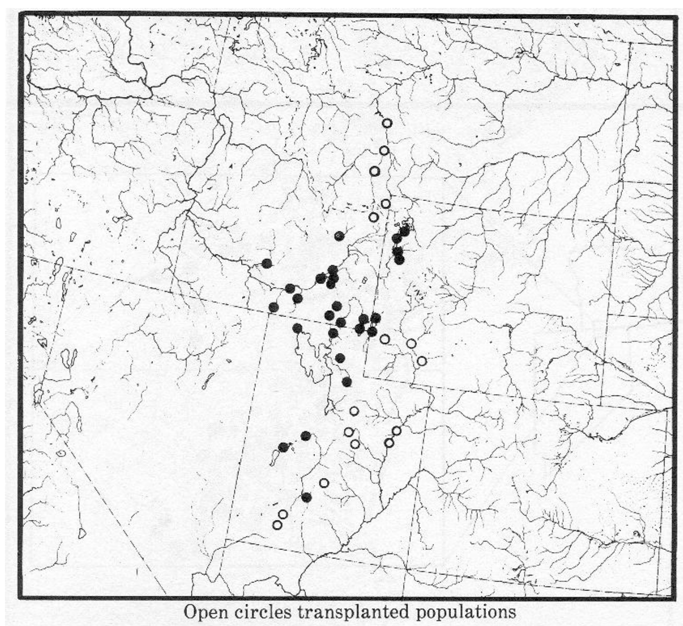


Figure 7. Native distribution of Utah chub.

Nonindigenous Occurrences: Figure 8 summarizes current distribution (native and introduced) projected from the dot map of Figure 7 which highlighted the species native distribution.

[7] Original material prepared by Pam Fuller and Leo Nico (revised March 20, 2001) at the Center for Aquatic Resource Studies . The Center is part of the Biological Resources Division of the Geological Survey within the U.S. Department of the Interior. Updated and edited, January 2004.

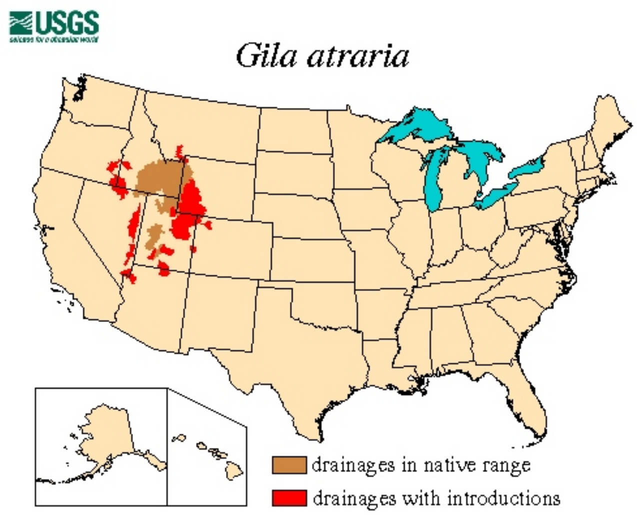


Figure 8. Native and introduced range of Utah chub.

The Utah chub is known from the Green and Yampa rivers in Colorado (Tyus et al. 1982). In Idaho, it has been recorded from the Snake River near Boise (circa 1970s) (Whitney and Wydoski 1979; Lee et al. 1980 et seq.), and Island Park Reservoir and several other small reservoirs in the upper part of Henrys Fork in the Snake River drainage (Simpson and Wallace 1978). The species was introduced into Hebgen Lake, Montana, where it became established and eventually spread to most of the Madison River and as far downstream as Canyon River Reservoir on the Missouri River (Brown 1971; Cross et al. 1986; Holton 1990), and it has been introduced into several sites in Nevada including Duck Lake (Lincoln County), Shoshone Springs (White Pine County), and both Murphy Spring and Comins Lake in Steptoe Valley (White Pine County) (Miller and Alcorn 1946; La Rivers 1962; Hubbs et al. 1974; Sigler and Sigler 1987; Miller et al. 1991). In Oregon, it was introduced to the Owyhee River system (Snake River drainage) (Bond 1994), and in Utah, Gila chub is has been collected from Panguitch Lake in the Sevier drainage, and from Fish Lake and reservoirs and streams in the Colorado River drainage (Sigler and Miller 1963; Vanicek et al. 1970; Holden and Stalnaker 1975b; Lee et al. 1980 et seq.; Tyus et al. 1982; Sigler and Sigler 1987). The species has also been recorded from the Green River in Wyoming (Baxter and Simon 1970; Lee et al. 1980 et seq.; Tyus et al. 1982; Hubert 1994).

Hubbs et al. (1974) detailed the introduction history of Utah chub in parts of the Great Basin of western North America. Tyus et al. (1982) gave a distribution map of this species in the upper Colorado basin. There is some uncertainty concerning the exact natural range of this species. For instance, Hubbs et al. (1974) stated that it might be theorized that an ancient outlet into the Bonneville system (where Utah chub is native), or some other stream connection, could have led to the spread of this species into Spring Valley; however, they noted that there is no definitive evidence of such a discharge, and no other indication of the occurrence of Bonneville fishes in Spring Valley. Most authors (e.g., Lee et al. 1980 et seq.; Sigler and Sigler 1987, 1996) have concluded that the Utah chub is native to the Snake River above Shoshone Falls but not below. Simpson and Wallace (1978) noted that the native range of Utah chub in Henrys Fork of the Snake River was restricted to below Mesa Falls; however, the fish was later introduced to Island park Reservoir and several small reservoirs in the area. Many western states have outlawed the use of live fish as bait to prevent the spread of this and other bait fish (Sigler and Sigler 1987).

Means of Introduction. Many introductions have been the result of bait bucket releases (Holden and Stalnaker 1975b; Simpson and Wallace 1978; Sigler and Sigler 1987). Hubbs et al. (1974) found evidence that the species may have been introduced to certain sites in the Great Basin by early Mormon settlers. These researchers also speculated that Native Americans may have brought Utah chubs into Shoshone Spring. The species may have been introduced to Murphy Spring, in Steptoe Valley, as forage for sportfish, although its establishment may have resulted from bait bucket releases (Hubbs et al. 1974). In some areas the species has become widespread because of natural dispersal from original points of introduction (e.g., Madison and Missouri rivers in Montana). Holden (1991) stated that it first appeared in Flaming Gorge Reservoir on the Wyoming-Utah border in 1964.

Status. Established in Colorado, Idaho, Nevada, Montana, Utah, Wyoming, and apparently Oregon. In its native range in the Green River and Yampa River in Colorado, and in the Dolores, Green, and Price Rivers in Utah, the species is considered rare or incidental. Utah chub is considered abundant in Flaming Gorge Reservoir in Utah and Wyoming (Tyus et al. 1982).

Impact of Introduction. Introduced populations often reach great abundance and become serious competitors with sport fish, especially trout (Sigler and Miller 1963). For instance, this species has been found to depress growth of kokanee salmon *Oncorhynchus nerka* through competition for food (Teuscher and Luecke 1996). Hubbs et al. (1974) also noted that it has a tendency toward population explosion and habitat dominance in artificial impoundments. Utah chub became a major management concern in Flaming Gorge Reservoir by the late 1960s, in part, because it appeared to compete with trout for planktonic foods and because the species established growth records of its own (Holden 1991). Introduced Utah chub, along with other introduced species, may have replaced the relict dace *Relictus solitarius* at Murphy Spring, Nevada (Hubbs et al. 1974). Predation by, and hybridization with, the Utah chub are considered some of the most serious hazards to the least chub *Iotichthys phlegethontis* in Utah (Sigler and Sigler 1987), a species proposed for federal listing as an endangered species (U.S. Fish and Wildlife Service 1997). Attempts at eradication have been largely unsuccessful and costly (Sigler and Sigler 1987).

References and Supplemental Bibliography

Anon., 2002 Fish collection database of the American Museum of Natural History. American Museum of Natural History, Central Park West , NY 10024-5192, USA.

Anon., 2001 Fish collection database of the National Museum of Natural History (Smithsonian Institution). Smithsonian Institution - Division of Fishes.

Anon., 1999 Fish collection database of the Natural History Museum, London (formerly British Museum of Natural History (BMNH)). Natural History Museum, London (formerly British Museum of Natural History (BMNH)).

Anon., 1996 Fish collection database of the University of British Columbia Fish Museum Fish Museum University of British Columbia, Vancouver, Canada.

Eschmeyer, W.N., Editor, 1999 Catalog of fishes. Updated database version of November 1999. Catalog databases as made available to FishBase in November 1999.

Eschmeyer, W.N., 1997 PISCES. Eschmeyer's PISCES database as published on the Internet in June 1997, URL: gopher://gopher.calacademy.org:640/7.

Hugg, D.O., 1996 MAPFISH georeferenced mapping database. Freshwater and estuarine fishes of North America. Life Science Software. Dennis O. and Steven Hugg, 1278 Turkey Point Road, Edgewater, Maryland, USA.

Hureau, J.-C., 1991 La base de donn,es GICIM: Gestion informatis,e des collections ichthyologiques du Mus,um. p. 225-227. In Atlas Preliminaire des Poissons d'Eaux Douce de France. Conseil Sup,rieur de la Pˆche, MinistŠre de l'Environment, CEMAGREF et Mus,um National d'Histoire Naturelle, Paris.

McAllister, D.E., 1990 A working list of fishes of the world. Copies available from D.E. McAllister, Canadian Museum of Nature, P.O. Box 3443, Ottawa, Ontario K1P 6P4, Canada. 2661 p. plus 1270 p. Index

Wu, H.L., K.T. Shao and C.F. Lai, Editors, 1999 Latin-Chinese dictionary of fishes names. The Sueichan Press, Taiwan.

Page, L.M. and B.M. Burr, 1991 A field guide to freshwater fishes of North America north of Mexico. Houghton Mifflin Company, Boston. 432 p. 368

Robins, C.R., R.M. Bailey, C.E. Bond, J.R. Brooker, E.A. Lachner, R.N. Lea and W.B. Scott, 1980 A list of common and scientific names of fishes from the United States and Canada. Am. Fish. Soc. Spec. Publ. (12)1-174.

Robins, C.R., R.M. Bailey, C.E. Bond, J.R. Brooker, E.A. Lachner, R.N. Lea and W.B. Scott, 1991 Common and scientific names of fishes from the United States and Canada. Am. Fish. Soc. Spec. Pub. (20):183 p.

[8]*Osmerus mordax* (Mitchill 1814)

Common Names: rainbow smelt

Taxonomy and identification: Scott and Crossman (1973); Becker (1983); Smith (1985); Page and Burr (1991).

Size: 33 cm

Native Range. Figure 9 illustrates the native range of the rainbow smelt. Atlantic drainages from Lake Melville, Newfoundland, to Delaware River, Pennsylvania, and west through Great Lakes; Arctic and Pacific drainages from Bathurst Inlet, Northwest Territories, to Vancouver Island, British Columbia. Also Pacific drainages of Asia (Page and Burr 1991).

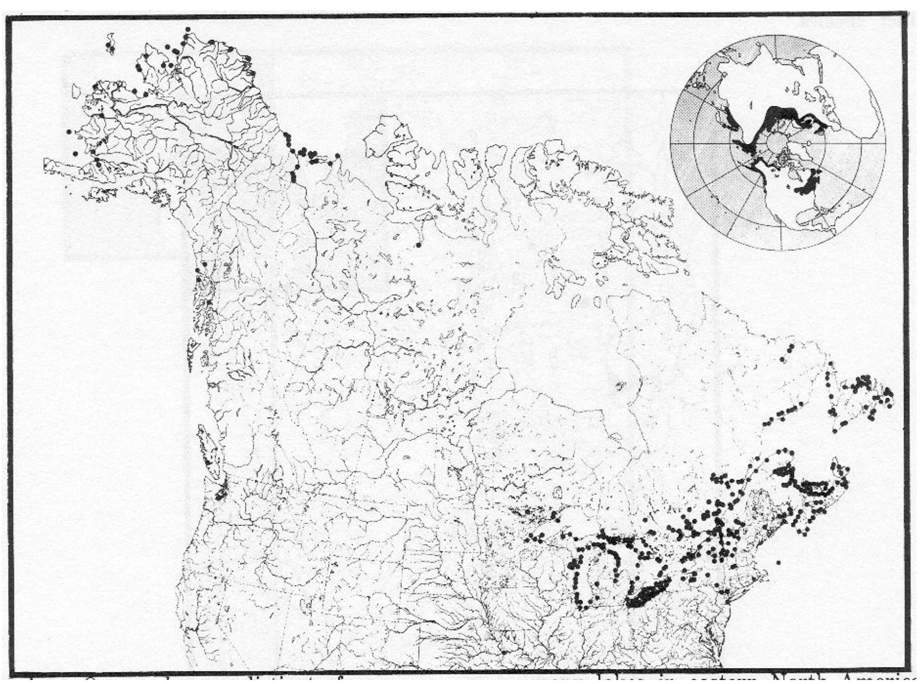


Figure 9. Native range of rainbow smelt (after Lee et al 1980).

[8]Original materials prepared by Pam Fuller (January 28, 2000) at the Center for Aquatic Resource Studies. The Center is part of the Biological Resources Division of the Geological Survey within the U.S. Department of the Interior. Updated and edited, January 2004.

Nonindigenous Occurrences: Figure 10 illustrates the distribution of rainbow smelt with introductions beyond the species' native range presented in the preceding dot map (Figure 9). Rainbow smelt occur in all five Great Lakes and also have been introduced or dispersed after introduction into several large rivers. Areas with introduced smelt include the Mississippi River,

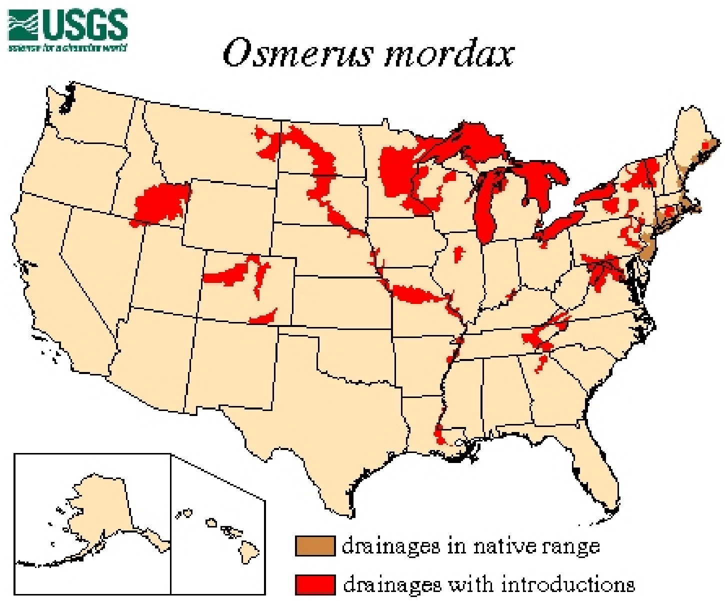


Figure 10. Expanded range (native plus introduced) of rainbow smelt.

Arkansas (Pennington et al. 1982; Mayden et al. 1987); reservoirs in the South Platte and Arkansas drainages and headwaters of the Colorado basin in Colorado (Woodling 1985; Propst and Carlson 1986); several lakes in Connecticut (Webster 1942); the Chattahoochee River below Lake Lanier, Georgia (Dahlberg and Scott 1971a, 1971b); lakes in the Sawtooth Mountains of Idaho (Linder 1963; Simpson and Wallace 1978); Lake Michigan (Emery 1985; Burr 1991), the Mississippi River, the Illinois River (Burr and Mayden 1980; Mayden et al. 1987; Burr 1991, Burr et al. 1996), and Ohio River (Burr 1991) in Illinois (Smith 1979; Burr and Page 1986); Lake Michigan and the Ohio River near Madison, Indiana (Emery 1985; Mayden et al. 1987); the Missouri River, Iowa (Harlan et al. 1987; Mayden et al. 1987); the Missouri River, Kansas (Mayden et al. 1987); the Mississippi River, Kentucky (Burr and Warren 1986; Mayden et al. 1987); the Mississippi River, Louisiana (Suttkus and Conner 1980; Mayden et al. 1987); Schoodic Lake, Maine (Havey 1973); Maryland (Ferguson 1876; Jenkins and Burkhead 1994); nonnative waters of Massachusetts (Smith 1833; Hartel 1992; Hartel et al. 1996); the Great Lakes in Michigan (Emery 1985); Lake Superior, Minnesota (Emery 1985; Burr and Page 1986); the

Missouri and Mississippi rivers, Missouri (Cross et al. 1986; Mayden et al. 1987; Pflieger 1997); the Missouri and Yellowstone rivers, Montana (Gould 1981; Cross et al. 1986; Mayden et al. 1987; Holton 1990); the Missouri River in Nebraska (Cross et al. 1986; Bouc 1987; Mayden et al. 1987); several dozen lakes in New Hampshire (Scarola 1973); Lake Erie (Emery 1985), Lake Ontario, the Finger Lakes, the Adirondack lakes, Neversink Reservoir, and Lake Champlain in New York (Werner 1980); Tennessee drainage, North Carolina (Menhinick 1991); Lake Sakakawea, North Dakota (Gould 1981; Bouc 1987; Harlan et al. 1987; Mayden et al. 1987; Holton 1990); Lake Erie, Ohio (Emery 1985); Lake Erie (Emery 1985) and Harvey's Lake (Susquehanna drainage), Pennsylvania (Denoncourt et al. 1975a; Hendricks et al. 1979; Cooper 1983; Hocutt et al. 1986); reservoirs on the Missouri River, South Dakota (Mayden et al. 1987; Hanten, personal communication); the Mississippi River, Watauga Reservoir, and South Fork Holston River, Tennessee (Mayden et al. 1987; Etnier and Starnes 1993); Lake Champlain, Vermont (Werner 1980); the Potomac River and Occoquan Reservoir, Virginia (Hocutt et al. 1986; Jenkins and Burkhead 1994); and Lake Superior, Wisconsin (Emery 1985; Burr and Page 1986).

Means of Introduction: The earliest known record is from 1912, when eggs were stocked in Crystal Lake, Michigan, which drains into Lake Michigan (Van Oosten 1937). Fish escaped into Lake Michigan and spread quickly throughout the Great Lakes and their tributaries (Creaser 1926; Gerking 1945; Hubbs and Lagler 1947; Nelson and Gerking 1968; Christie 1974; Eddy and Underhill 1974; Smith 1979; Morrow 1980; Phillips et al. 1982; Cooper 1983; Emery 1985). Early records documenting the smelt's range expansion in the Great Lakes include Lake Michigan, 1923 (Christie 1974; Emery 1985), Lake Erie, 1935 (Cooper 1983; Smith 1985), Lake Huron, 1925 (Christie 1974; Eddy and Underhill 1974), Lake Ontario, 1929 (Christie 1974; Smith 1985), and Lake Superior, 1923 (Emery 1985). The Lake Ontario population may be either native to this lake or gained access when the Welland Canal was built (Emery 1985; Smith 1985). Another possibility is that the species was introduced from the Finger Lakes via the Seneca-Cayuga, Erie and Oswego canals (Smith 1985).

Two means have been proposed to explain the introduction of rainbow smelt into the Missouri and Mississippi rivers. It may have spread from Lake Michigan via the Chicago sanitary canal to the Illinois River and then to the Mississippi and Missouri rivers (Burr and Mayden 1980). Alternatively, the species may have gained access to these rivers as a result of a stocking at Lake Sakakawea, North Dakota, in 1971 (Bouc 1987; Mayden et al. 1987; Holton 1990). The second explanation seems more plausible because of a lack of records from the Illinois River. Records of first occurrence in other areas include the Mississippi River, Illinois and Kentucky, 1978; Mississippi River, Louisiana, 1979; Mississippi River, Tennessee and Arkansas, 1980; Missouri River, Missouri, 1980; Missouri River, Kansas, 1982 (Mayden et al. 1987). Mayden et al. (1987) provided a map of the species' distribution, dates of first observation in new areas, and possible introduction pathways. The species was originally introduced into Lake Sakakawea, North Dakota, as forage for salmonids (Mayden et al. 1987).

Status: Introduced populations of rainbow smelt have been very successful and the species is now established in the Great Lakes and in most rivers and lakes where introduced. In the Great Lakes a commercial fishery targeting smelt has been operating for many years (Smith 1985), and it is the most abundant fish in some locations on the Mississippi River (Pflieger 1997). To date, no adults of the rainbow smelt have been found in either Tennessee (Etnier and Starnes 1993) or Missouri (Pflieger 1997). The latter observation leads Pflieger (1997) to conclude that fish collected in Missouri are probabily maintained by continued escape of fish from upstream reservoirs on the Missouri River. As of 1987, only one specimen had been taken from the Ohio River (Mayden et al. 1987), and rainbow smelt are considered extirpated in Georgia, since none have been collected since its original release (Dahlberg and Scott 1971b).

Impact of Introduction: In the Great Lakes, rainbow smelt compete with lake herring *Coregonus artedii* for food (Becker 1983). In early studies, Havey (1973) had reported increased growth of landlocked Atlantic salmon following the introduction of smelt as a forage species in a lake in Maine, and Christie (1974) correlated lake herring decline with smelt increases in most of the lakes. Todd (1986b) also reported that smelt may be partially responsible for the decline of whitefish (*Coregonus* spp.) in the Great Lakes. Hrabik et al. (1998) found evidence of competition for food between introduced rainbow smelt and native yellow perch *Perca flavescens* in Wisconsin lake habitats. Rainbow smelt are eaten by humans and used as bait for salmonids and walleye (Pflieger 1997).

References

Becker, G. C. 1983. Fishes of Wisconsin. University of Wisconsin Press, Madison, WI.

Bouc, K. 1987. The fish book. Nebraskaland Magazine 65(1):1-130.

Burr, B. M., and R. L. Mayden. 1980. Dispersal of rainbow smelt, Osmerus mordax, into the upper Mississippi River (Pisces:Osmeridae). American Midland Naturalist 104(1):198-201.

Burr, B. M., and M. L. Warren, Jr. 1986. A distributional atlas of Kentucky fishes. Kentucky Nature Preserves Commission Scientific and Technical Series 4. 398 pp.

Cooper, E. L. 1983. Fishes of Pennsylvania. Pennsylvania State University Press, University Park, PA.

Creaser, C. W. 1926. The establishment of the Atlantic smelt in the upper waters of the Great Lakes. Papers of the Michigan Academy of Science, Arts, and Letters 5(1925):405-424.

Dahlberg, M. D., and D. C. Scott. 1971a. The freshwater fishes of Georgia. Bulletin of the Georgia Academy of Science 29:1-64.

Dahlberg, M. D., and D. C. Scott. 1971b. Introductions of freshwater fishes in Georgia. Bulletin of the Georgia Academy of Science 29:245-252.

Denoncourt, R. F., T. B. Robbins, and R. Hesser. 1975a. Recent introductions and reintroductions to the Pennsylvania fish fauna of the Susquehanna River drainage above Conowingo Dam. Proceedings of the Pennsylvania Academy of Science 49:57-58.

Eddy, S., and J. C. Underhill. 1974. Northern fishes, with special reference to the Upper Mississippi Valley, 3rd edition. University of Minnesota Press, Minneapolis, MN.

Emery, L. 1985. Review of fish introduced into the Great Lakes, 1819-1974. Great Lakes Fishery Commission Technical Report, volume 45. 31 pp.

Etnier, D. A., and W. C. Starnes. 1993. The fishes of Tennessee. University of Tennessee Press, Knoxville, TN.

Ferguson, T. B. 1876. Report of the Commissioners of Fisheries of Maryland to the General Assembly. 1 January 1876. John F. Wiley, Annapolis, MD.

Gerking, S. D. 1945. Distribution of the fishes of Indiana. Investigations of Indiana Lakes and Streams 3:1-137.

Hanten, R.L. - Department. of Game, Fish, and Parks, Pierre, SD. Response to NBS-G nonindigenous questionaire.

Harlan, J. R., E. B. Speaker, and J. Mayhew. 1987. Iowa fish and fishing. Iowa Department of Natural Resources, Des Moines, IA. 323 pp.

Hartel, K. E. 1992. Non-native fishes known from Massachusetts freshwaters. Occasional Reports of the Museum of Comparative Zoology, Harvard University, Fish Department, Cambridge, MA. 2. September. pp. 1-9.

Hendricks, M. L., J. R. Stauffer, Jr., C. H. Hocutt, and C. R. Gilbert. 1979. A preliminary checklist of the fishes of the Youghiogheny River. Chicago Academy of Sciences, Natural History Miscellanea 203:1-15.

Holton, G. D. 1990. A field guide to Montana fishes. Montana Department of Fish, Wildlife and Parks, Helena, MT. 104 pp.

Hubbs, C. L., and K. F. Lagler. 1949. Fishes of Isle Royale, Lake Superior, Michigan. Reprinted from Papers of the Michigan Academy of Science, Arts, and Letters, volume XXXIII, 1947. Contributions from the Scripps Institute of Oceanography, New Series, volume 420.

Jenkins, R. E., and N. M. Burkhead. 1994. Freshwater fishes of Virginia. American Fisheries Society, Bethesda, MD.

Linder, A. D. 1963. Idaho's alien fishes. Tebiwa 6(2):12-15.

Mayden, R. L., F. B. Cross, and W. T. Gorman. 1987. Distributional history of the rainbow smelt, *Osmerus mordax* (Salmoniformes: Osmeridae), in the Mississippi River Basin. Copeia 1987(4):1051-1054.

Menhinick, E. F. 1991. The freshwater fishes of North Carolina. North Carolina Wildlife Resources Commission. 227 pp.

Morrow, J. E. 1980. The freshwater fishes of Alaska. Alaska Northwest Publishing Company, Anchorage, AK.

Nelson, J. S., and S. D. Gerking. 1968. Annotated key to the fishes of Indiana. Project 342-303-815. Department of Zoology, Indiana Aquatic Research Unit, Indiana State University, Bloomington, IN.

Page, L. M., and B. M. Burr. 1991. A field guide to freshwater fishes of North America north of Mexico. The Peterson Field Guide Series, volume 42. Houghton Mifflin Company, Boston, MA.

Pennington, C. H., G. J. Dahl, and H. L. Schramm, Jr. 1982. Occurrence of the rainbow smelt in the lower Mississippi River. Journal of the Mississippi Academy of Sciences 27:41-42.

Phillips, G. L., W. D. Schmid, J. C. Underhill. 1982. Fishes of the Minnesota region. University of Minnesota Press, Minneapolis, MN.

Scarola, J. F. 1973. Freshwater fishes of New Hampshire. New Hampshire Fish and Game Department, Division of Inland and Marine Fisheries. 131 pp.

Simpson, J., and R. Wallace. 1978. Fishes of Idaho. University of Idaho Press, Moscow, ID.

Smith, C. L. 1985. The inland fishes of New York state. New York State Department of Environmental Conservation, Albany, NY. 522 pp.

Smith, J. V. C. 1833. A natural history of the fishes of Massachusetts. A reprint of the 1833 edition. Freshet Press, Inc., Rockville Center, NY.

Smith, P. W. 1979. The fishes of Illinois. University of Illinois Press, Urbana, IL.

Suttkus, R. D., and J. V. Conner. 1980. The rainbow smelt, *Osmerus mordax*, in the lower Mississippi River near St. Francisville, Louisiana. American Midland Naturalist 104(2):394.

Todd, T. N. 1986b. Artificial propagation of coregonines in the management of the Laurentian Great Lakes. Arch. Hydrobiol. Beih./Ergebn. Limnol. 22:31-50.

Van Oosten, J. 1937. The dispersal of smelt, *Osmerus mordax* (Mitchill), in the Great Lakes region. Transactions of the American Fisheries Society 66:16-171.

Webster, D. A. 1942. The life histories of some Connecticut fishes. Pages 122-227 in State Board of Fisheries and Game. A fishery survey of important Connecticut lakes. Connecticut Geological and Natural History Survey 63.

Werner, R. G. 1980. Freshwater fishes of New York state: a field guide. Syracuse University Press, Syracuse, NY.

Woodling, J. 1985. Colorado's little fish: a guide to the minnows and other lesser known fishes in the state of Colorado. Colorado Division of Wildlife, Denver, CO. 77 pp.

Asian Carp

Asian carp are large fish (39-40 inch; 40-50 lb.) introduced into the U.S. by fish farmers in Southern states in the 1960's and 70's to control vegetation and algae blooms. Three of these species, bighead carp (*Hypophthalmichthys nobilis*), the grass carp (*Ctenopharyngodon idella*), and silver carp (*Hypophthalmichthys molitrix*) have been released or have escaped to the wild and are reproducing in many rivers and streams of the Mississippi River Basin. Black carp (*Mylopharyngodon piceus*) has also been included, since it remains in captivity in hatcheries, fish culture facilities, and fish farm ponds, primarily in Southeastern states. Common carp (*Cyprinus carpio*) has not been considered as part of this compilation.

[9]***Hypophthalmichthys (Aristichthys) nobilis*** (Richardson, 1845)

Common name: Bighead carp

Size: Maximum length generally falls between 100 to 150 cm TL and maximum weight varies from 20 to 50 kg.

Native distribution. *Hypophthalmichthys (Aristichthys) nobilis* is one of many carp species whose native distribution occurs in Asia, being particularly common to large rivers of eastern China (Figure 11).

Taxonomy and identification. Bighead carp are dark green to olive in color on their backs with gray to silvery sides and a white to cream colored belly, generally having small black blotches over their dorsolateral surfaces. Bighead carp lack barbels and are distinctive morphologically, having scales that are very tiny and eyes are set below the midline of the long and compressed body.

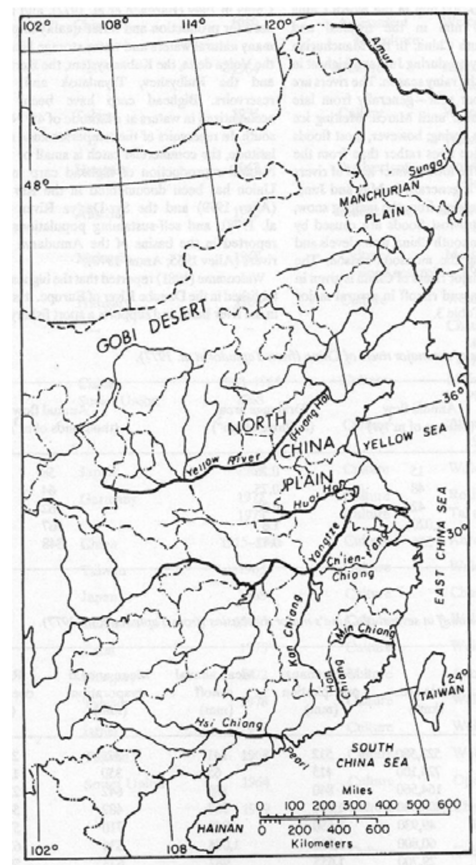


Figure 11. Native ranges of Asian carp occur in China.

[9]Original material prepared by Pam Fuller, USGS/BRD/FISC, with additional material obtained from FishBase (www.fishbase.org), access October through December, 2003. Updated and edited, November, 2004.

The head is very large compared to the body (hence, the species common name), and the fish can weigh from 55-110 lbs (25-50 kg) and measure nearly 60 inches (1.5 meters) long. The species is technically characterized by spine counts: dorsal spines (total): 3-3; dorsal soft rays (total): 7-7; anal spines: 1-3; anal soft rays: 12-14. The posterior margin of the last simple dorsal ray is not serrated, and branched anal rays number 13-14.5. A keel extend from base of the pelvis to anus.

Life history. Bighead carp are freshwater benthopelagic fishes that generally range between near surface to 5 m. Their apparent perferred temperatures range between 4 - 26˚C which are common to their native and introduced ranges that lie between latitude 64ºN to 18ºS. Populations of bighead carp are relatively resilient to adverse impacts, having a minimum population doubling time that ranges between 1.4 and 4.4 years. Preferred habitats include rivers and lakes where the fish feeds on the bottom, mainly on zooplankton. In its native range, the fish is marketed fresh and frozen.

Bighead carp have long gill rakers, which allow them to strain plankton from the water for food, which includes cyanobacteria, green algae, zooplankton, and aquatic insects and larva. The fish forage constantly, and are voracious feeders. Bighead carp will compete for food with fish that are still in the larval stage and fish populations decrease because the larval fish do not get enough food to survive.

Bighead carp spawn when the water temperature is between 25 to 30 ºC (77 to 86 ºF) which means active spawning at mid-latitudes occurs between April and June. Peak spawning season occurs in late May, with females carrying between 660,000 to 872,000 eggs. The number of eggs lain increases with age of female until senescence when production decreases.

Nonindigenous occurrences: The bighead carp was introduced to the Mississippi River when private hatchery ponds were washed out in the state of Arkansas in the 1970's or possibly they were let go into the wild when they were no longer needed on by the fish farmers. Fish appeared in open water in the early 1980's in the Ohio and Mississippi Rivers, and at present bighead carp have been found in at least 19 states including Lake Erie. Introductions of *Hypoopthalmichthys (Aristichthys) nobilis* have been documented worldwide [FishBase cites, n=71], ranging from locations near native distribution (e.g., China to Malaysia throughout the 1800's, and from China to Japan, southeast Asia from the end of the 19th through the 20th century) to releases throughout Europe in mid- to late 20th century that yielded sustained populations in eastern Europe. These eastern European populations in turn served as source regions for invasions, most unsuccessful, into western Europe. Current distribution of commercial stock is worldwide, and releases from captivity regularly occur although establishment of sustainable populations is infrequent. The earliest documentation of release and establishment of sustainable populations in North America occurred in the mid-1980's (FishBase cites 1986), which resulted from ornamental fish being released from confinement. FishBase BiOSC cited point data for *Hypophthalmichthyes nobilis* (*Aristichthys nobilis*; n = 7) includes type specimens at University of Kansas (*Hypophthalmichthys nobilis*, 1993 at lat 38.98, long -96.77 (Catalog KU 23151), collected by T. Hall near Kansas River below Bowersock Dam at Lawrence). In North America, the increasing range of *Hypophthalmichthys nobilis* is apparent with increased reports as yet to be cataloged by museums and subscribers to BiOSC. Each citing in North America to date is compatible with the species current distribution (Figure 12).

Methods of introduction: Most introductions have been intentional, since the bighead carp

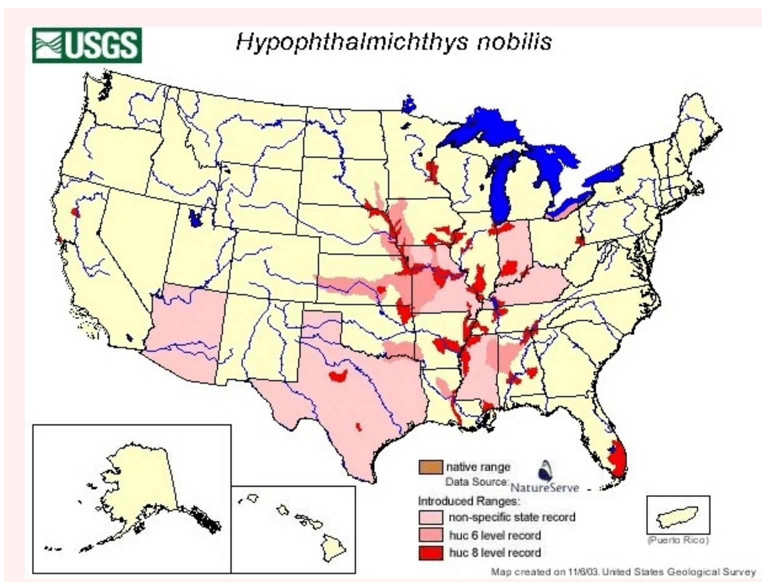


Figure 12. Current extent of bighead carp in the US.

supports a highly successful commercial fishery and aquaculture industry (e.g., commercial aquaria and show aquaria). Introduced populations occur in numerous countries, and the species has achieved a near global distribution. Many introductions, however, have not yielded sustainable populations outside culture facilities, since the species' breeding requirements are very specialized and commerical stocks are generally maintained by artificial reproduction or continuous importation. When the species does escape from captivity, several countries report adverse ecological impact after bighead carp are introduction to a region and establish sustainable populations.

Impacts. Bighead carp is not included on the IUCN Red List, and is considered a potential pest. A number of diseases associated with *Hypophthalmichthys nobilis* have been observed and potentially threaten other carp and native cyprinids, if disease agents can jump from one species to another. These diseases include parasitic infections: parasitic protozoa and worms (such as White Spot Disease, Dactylogyrus Gill Flukes Disease, Trichodinosis, Cryptobia Infestation, Turbidity of the skin in freshwater fish, Myxidium Infection, Trichodina Infection 1, Trichodina Infection 2, Trichodina Infection 3, Tripartiella Infestation, Trichodinella Infection 2, Ichthyobodo Infection, Bothriocephalus Infestation), fungal disease (e.g., False Fungal Infection caused by *Apiosoma* spp.), and Fish Louse Infestation 2.

Methods of control. Currently, the best control technology is far from perfect, and currently it appears that big head carp are restricted only by high dams and electric barriers (also see, e.g., http://www.umesc.usgs.gov/invasive_species/asian_carp.html, http://www.glfc.org/fishmgmt/carp.asp, and http://www.epa.gov/glnpo/invasive/asiancarp/).

References and Supplemental Bibliography (derived from FishBase; http://www.fishbase.org/search.cfm)

Abdusamadov, A.S., 1986 Biology of White Amur, Ctenopharyngodon idella, silver carp, Hypophthalmichthys molitrix, and bighead, Aristichthys nobilis, acclimatized in the Terek region of the Caspian basin. J. Ichthyol. 26(4):41-49.

Anon., 2001, Fish collection database of the National Museum of Natural History (Smithsonian Institution). Smithsonian Institution - Division of Fishes.

Anon., 2000. Fish collection database of the Gulf Coast Research Laboratory (GCRL) The Gulf Coast Research Laboratory (GCRL), Ocean Springs, Mississippi, USA.

Anon., 2003 Fish Collection of the Royal Ontario Museum Royal Ontario Museum

Anon., 1994 Atlas pesquero de México. Instituto Nacional de la Pesca. 234 p.

Anon., 2002, Fish collection database of the American Museum of Natural History American

Museum of Natural History, Central Park West at 79th Street, New York, NY 10024-5192, USA

Anon., 1999, Fish collection database of the Natural History Museum, London (formerly British Museum of Natural History (BMNH)). Natural History Museum, London (formerly British Museum of Natural History (BMNH)).

Cremer, M. C. and R. O. Smitherman, 1980 Food habits and growth of silver and bighead carp in cages and ponds. Aquaculture 20:57-64

Cui, J., X, Ren and Yu, Q., 1991 Nuclear DNA content variation in fishes. Cytologia 56:425-429.

Eschmeyer, W.N., 1990 Catalog of the genera of recent fishes. California Academy of Sciences, San Francisco, USA. 697 p.

Eschmeyer, W.N., 1997 PISCES. Eschmeyer's PISCES database as published on the Internet in June 1997, URL: gopher://gopher.calacademy.org:640/7.

FAO Fishery Information, Data and Statistics Service, 1993 Aquaculture production (1985-1991). FAO Fish. Circ. 815, Rev. 5. 213 p.

FAO, 1997 Aquaculture production statistics 1986-1995. FAO Fish. Circ. 815, Rev. 9. 195 p.

FAO, 1997 FAO Database on Introduced Aquatic Species. FAO Database on Introduced Aquatic Species, FAO, Rome.

FAO Fishery Information, Data and Statistics Service, 1991 Aquaculture production (1986-1989). FAO Fish. Circ. 815, Rev. 3. 141 p.

FAO, 2000 Fishery statistics: Capture production. FAO Fish. Ser. No. 54. FAO Statistics Series No. 152. 713 p.

FAO, 1992 FAO Yearbook. Fishery statistics: catches and landings, volume 74. FAO Fish. Series 43. 677 p.

FAO-FIDI, 1997International Standard Statistical Classification of Aquatic Animals and Plants

(ISSCAAP). November 1997. Fishery Information, Data and Statistics Service, Fisheries Department, FAO, Rome, Italy.

Garibaldi, L., 1996 List of animal species used in aquaculture. FAO Fish. Circ. 914. 38 p. 12228, FAO, 1996 Aquaculture production statistics 1985-1994. FAO Fish. Circ. 815. 189 p.

Holcík, J., 1991Fish introductions in Europe with particular reference to its central and eastern part. Can. J. Fish. Aquat. Sci. 48 (Suppl. 1):13-23.

IGFA, 2001 Database of IGFA angling records until 2001. IGFA, Fort Lauderdale, USA.

Kottelat, M., 1997 European freshwater fishes. Biologia 52, Suppl. 5:1-271.

Lever, C., 1996 Naturalized fishes of the world. Academic Press, California, USA. 408 p.

Li, S., B. Zhou and Q. Lin, 1990 The yield and growth of major fish species in a large Chinese reservoir. Asian Fish. Sci. 3(2):185-196.

Manea, G., 1985 Acclimatization of new fishes and other aquatic organisms. Ceres Publishing House, Bucharest.

Nichols, J.T., 1943. The freshwater fishes of China. Natural history of Central Asia: Volume IX. The American Museum of Natural History, New York, USA, 322 p.

Ren, X., J. Cui, and Q. Yu, 1992 Chromosomal heterochromatin differentiation of cyprinid fishes. Genetica 87:47-51

Robins, C.R., R.M. Bailey, C.E. Bond, J.R. Brooker, E.A. Lachner, R.N. Lea and W.B. Scott, 1991, World fishes important to North Americans. Exclusive of species from the continental waters of the United States and Canada. Am. Fish. Soc. Spec. Publ. (21):243 p.

Robins, C.R., R.M. Bailey, C.E. Bond, J.R. Brooker, E.A. Lachner, R.N. Lea and W.B. Scott, 1991 Common and scientific names of fishes from the United States and Canada. Am. Fish. Soc. Spec. Pub. (20):183 p.

Welcomme, R.L., 1988 International introductions of inland aquatic species. FAO Fish. Tech. Pap. No. 294. 318 p.

Zan, R. and Z. Song, 1980 Comparative analysis of karyotypes of Cyprinus carpio and Carassius auratus, as well Aristichthys nobilis and Hypophthalmichthys molitrix. Acta genet. sin. 7(1):72-76.

[10]***Hypophthalmichthys molitrix*** (Valenciennes 1844)

Common Name: silver carp

Size: Silver carp easily reach a maximum size of 100 cm TL and 27 kg with a published maximum size of 105 cm TL (male/unsexed) and 50.0 kg.

Taxonomy and identification. Silver carp are olivaceous to silvery in color and lack barbels. Distinguishing characteristics were initially summarized by Berg (1949), and currently published state and regional fish books provided detailed accounts of life history (e.g., Robison and Buchanan 1988; Etnier and Starnes 1993; Pflieger 1997). Diagnostically, the species presents spine counts: dorsal spines (total): 1-3; dorsal soft rays (total): 6-7; anal spines: 1-3; anal soft rays: 10-14 (branched 12-13.5). The edge of last simple dorsal ray is not serrated, and a keel extends from isthmus to anus.

Life history: *Hypopthalmichthyes moltrix* is requires standing or slow-flowing conditions such as in impoundments or the backwaters of large rivers, so tends to be benthopelagic and potamodromous and ranges between surface to 5m in depth. As one of the so-called "Chinese carps" or "Asian carps," the silver carp is a filter-feeder capable of taking large amounts of phytoplankton, zooplankton, bacteria, and detritus (Leventer 1987). In its native range, silver carp migrate upstream to breed, then fertilized eggs and larvae float downstream to floodplain zones. The species generally swims just beneath the water's surface, and is very active swimmer being well known for its habit of leaping clear of the water when disturbed.

In its native range (approximately bounded between 64ºN and 43ºS), temperatures range from 6 to 28ºC. As is characteristic of other "Asian carp," *Hypopthalmichthyes moltrix* is relative resilience and has a population doubling time that ranges between 1.4 to 4.4 years.

Native Range. Several major Pacific drainages in eastern Asia from the Amur River of far eastern Russia (Siberia) south through much of eastern half of China to Pearl River, possibly including northern Vietnam (Berg 1949; Li and Fang 1990).

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[10]Original material prepared by Leo Nico (October 3, 2003) at the USGS/BRD/FISC. Supplemental materials were also incorporated from FishBase, then expanded and edited, October, 2003 through November, 2004.

Nonindigenous Occurrences: Silver carp have been introduced around the world for aquaculture and control of algal blooms. In North America (Figure 13), the species has been recorded from the Black Warrior and Tallapoosa river drainages of the Mobile Basin, including Yates Reservoir, in Alabama (Mettee et al. 1996; J. Hornsby and M. Pierson, personal communication); from

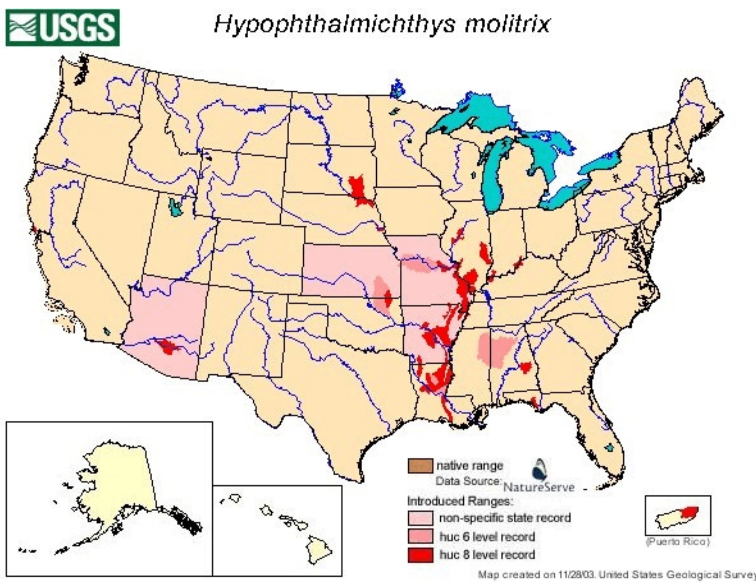

Figure 13. Current distribution of silver carp in US.

Arizona (Courtenay et al. 1991; Rinne 1995); and from the Arkansas and White River systems (including the the lower Cache River) and the Mississippi River mainstem in Arkansas (Freeze and Henderson 1982; Carter and Beadles 1983; Courtenay et al. 1984; Robison and Buchanan 1988). It has been stocked in water treatment ponds on the East Slope of Colorado (D. Horak, personal communication). A single specimen was taken in August 1994 from St. Andrews Bay at the Deer Point Lake spillway, Bay County, Florida (Middlemas 1994). It has been intentionally released in Hawaii (Davidson et al. 1992). It has also been collected or reported from several water bodies in, or bordering, Illinois, including the Mississippi and Ohio rivers and several of their tributaries, the Muddy River, Horseshoe Lake and vicinity in the Cache River drainage (Burr 1991; Burr et al. 1996; Laird and Page 1996) and the Embarras River below Lake Charleston (K. Cummings, personal communication). There are also records of this species from the southeastern part of Indiana (presumably the Ohio River; Courtenay et al. 1991; Simon et al. 1992); unspecified location(s) in Kansas (Courtenay et al. 1991) (possibly the Missouri River); from the Ohio River in Kentucky (Pearson and Krumholz 1984; Burr and Warren 1986); from the lower Mississippi River and many tributary sites in Louisiana including the Atchafalaya, Red, Boeuf, Old, Ouachita, and Little river drainages, LaFourche Canal, Miller Lake, and Loggy Bayou (Freeze and

Henderson 1982; Carp Task Force 1989; Douglas et al. 1996; F. Bryan and J. Hughes Little, personal communication); from the Mississippi and Missouri river mainstems in Missouri (Courtenay et al. 1991; Robinson 1995; Pflieger 1997); the Missouri River drainage and Elkhorn River Nebraska (Nebraska Game and Parks 2000); the Missouri River up to Gavins Point Dam in South Dakota (W. Stancill, pers. comm.), the mouth of the James River in South Dakota (R. Klumb, pers. comm.)and from a Mississippi River outflow in Tennessee (C. Saylor, personal communication). Voucher specimens are housed state museums in Florida (UF 98162); Illinois (SIUC 17716, 23043, 23046, 24415; INHS 88425); Louisiana (NLU 65811, 66858, 66859).

Means of introduction and status. Introductions of silver carp have historically been intentional, stemming from species' commercial fishery and aquaculture value. When released or escaped from culture, and sustained populations are subsequently attained, the species is frequently characterized as a potential pest. FishBase cites no fewer than 91 introductions of *Hypophthalmichthys molitrix* world-wide, which suggests the species is highly adaptable with life history attributes amenable to an invasive (see http://www.fishbase.org/search.cfm). Initial invasion and establishment in US poorly documented but the species has been imported and stocked for phytoplankton control in eutrophic water bodies. Silver carp have also been stocked as a food fish, e.g., the species was first brought into the United States in 1973 when a private fish farmer imported silver carp into Arkansas (Freeze and Henderson 1982). By the mid-1970's the silver carp was being raised at six state, federal, and private facilities, and by the late 1970s the species had been stocked in several municipal sewage lagoons (Robison and Buchanan 1988). By 1980 the species was discovered in natural waters, probably a result of escapes from fish hatcheries and other types of aquaculture facilities (Freeze and Henderson 1982). The occurrence of silver carp in the Ouachita River of the Red River system in Louisiana was likely the result of an escape from an aquaculture facility upstream in Arkansas (Freeze and Henderson 1982). The Florida introduction was probably a result of stock contamination, a silver carp having been inadvertently released with a stock of grass carp being used for aquatic plant control (Middlemas 1994). In a similar case, the species was apparently introduced accidentally to an Arizona lake as part of an intentional, albeit illegal, stock of diploid grass carp (W. Silvey, personal communication). Pearson and Krumholz (1984) suggested that individuals taken from the Ohio River may have come from plantings in local ponds or entered the Ohio River from populations originally introduced in Arkansas.

Impact of Introduction. Silver carp have been intensively cultured in many parts of the world, often raised in combination with other fishes, and the species is among 3 or 4 species of cyprinids whose world production in aquaculture exceed 1 million tons/year, much as fresh fish for human consumption. The species has also been introduced to many countries where its ability to clean reservoirs and other waters of clogging algae is exploited, e.g., Jenkins and Burkhead (1994) reported on the use of silver carp in a wastewater treatment pond in the upper James River drainage of Virginia, yet no record of the species in Virginia open waters has occurred. Pflieger (1997) considered the impact of this species difficult to predict because of its place in the food web. In numbers, the silver carp has the potential to cause enormous damage to native species because it feeds on plankton required by larval fish and native mussels (Laird and Page 1996). Silver carp are also potential competitors with adults of some native fishes (e.g., gizzard shad) that also rely on plankton for food (Pflieger 1997).

Adverse ecological impacts have been observed after the species' introduction, e.g., increased incidence of fish disease associated with *Hypophthalmichthys molitrix* such as parasitic protozoa and parasitic worms (Bothriocephalus Infestation 2, Myxobolus Infection 4, Myxobolus Infection 2, Anchorworm Disease (Lernaea sp.), Dactylogyrus Gill Flukes Disease, and Trichodinosis) and bacterial diseases (e.g., enteric redmouth).

References and Supplemental Bibliography (derived from FishBase)

Anon., 2003 Fish collection of the Royal Ontario Museum. Royal Ontario Museum.

Anon., 1996 Fish collection database of the University of British Columbia Fish Museum Fish Museum University of British Columbia, Vancouver, Canada.

Anon., 2002 Fish collection database of the American Museum of Natural History. American Museum of Natural History, Central Park West , NY 10024-5192, USA.

Anon., 1999 Fish collection database of the Natural History Museum, London (formerly British Museum of Natural History (BMNH)). Natural History Museum, London (formerly British Museum of Natural History (BMNH)).

Anon., 2001 Fish collection database of the National Museum of Natural History (Smithsonian Institution). Smithsonian Institution - Division of Fishes.

Anon., 1994 Atlas pesquero de Mexico. Instituto Nacional de la Pesca. 234 p.

Brummett, R.E., R.O. Smitherman and R.A. Dunham., 1988 Isozyme expression in bighead carp, silver carp and their reciprocal hybrids. Aquaculture. 70:21-28.

Chakalall, B. (compiler), 1993 Species cultured in insular Caribbean countries, Belize, French Guiana, Guyana and Suriname. Caribbean Technical Co-operation Network in Artisanal Fisheries an Aquaculture. FAO Regional Office for Latin America and the Caribbean. RLAC/93/28-PES-24. Santiago, Chile. 32 p.

Courtenay, W.R. Jr., D.A. Hensley, J.N. Taylor and J.A. McCann, 1984 Distribution of exotic fishes in the continental United States. p. 41-77. In W.R. Courtenay, Jr. and J.R. Stauffer, Jr. (eds.) Distribution, biology and management of exotic fishes. Johns Hopkins University Press, Baltimore, USA.

Cremer, M. C. and R. O. Smitherman, 1980 Food habits and growth of silver and bighead carp in cages and ponds. Aquaculture 20:57-64

Cui, J., X, Ren and Yu, Q., 1991 Nuclear DNA content variation in fishes. Cytologia 56:425-429.

Eldredge, L.G., 1994 Freshwater fishes. p. 73-84. In L.G. Eldredge, Perspectives in aquatic exotic species management in the Pacific Islands. Vol. 1. Introductions of commercially significant aquatic organisms to the Pacific Islands. South Pacific Commission, New Caledonia.

Erdman, D.S., 1984 Exotic fishes in Puerto Rico. p. 162-176. In W.R. Courtney, Jr. and J.R. Stauffer, Jr. (eds.) Distribution, biology and management of exotic fishes. Johns Hopkins University Press, Baltimore, USA.

Eschmeyer, W.N., Editor, 1999 Catalog of fishes. Updated database version of November 1999. Catalog databases as made available to FishBase in November 1999.

FAO, 1997 FAO Database on Introduced Aquatic Species. FAO Database on Introduced Aquatic Species, FAO, Rome.

FAO Fishery Information, Data and Statistics Service, 1991 Aquaculture production (1986-1989). FAO Fish. Circ. 815, Rev. 3. 141 p.

FAO, 2000 Fishery statistics: Capture production. FAO Fish. Ser. No. 54. FAO Statistics Series No. 152. 713 p.

FAO, 1996 Aquaculture production statistics 1985-1994. FAO Fish. Circ. 815. 189 p.

Garibaldi, L., 1996 List of animal species used in aquaculture. FAO Fish. Circ. 914. 38 p.

Holcik, J., 1991 Fish introductions in Europe with particular reference to its central and eastern part. Can. J. Fish. Aquat. Sci. 48 (Suppl. 1):13-23.

Horne, M.T. and A.C. Barnes, 1999 Enteric redmouth disease (Yersinia ruckeri). p.455-477. In P.T.K. Woo and D.W. Bruno (eds.) Fish Diseases and Disorders, Vol. 3: Viral, Bacterial and Fungal Infections. CAB Int'l.

Houde, E.D. and C.E. Zastrow, 1993 Ecosystem- and taxon-specific dynamic and energetics properties of fish larvae assemblages. Bull. Mar. Sci. 53(2):290-335.

IGFA, 2001 Database of IGFA angling records until 2001. IGFA, Fort Lauderdale, USA.

Klinkhardt, M, M. Tesche and H. Greven, 1995 Database of fish chromosomes. Westarp Wissenschaften.

Kottelat, M., 1997 European freshwater fishes. Biologia 52, Suppl. 5:1-271.

Lazzaro, X., 1987 A review of planktivorous fishes: their evolution, feeding behaviours, selectivities, and impacts. Hydrobiologia 146:97-167.

Lever, C., 1996 Naturalized fishes of the world. Academic Press, California, USA. 408 p.

Li, S., B. Zhou and Q. Lin, 1990 The yield and growth of major fish species in a large Chinese reservoir. Asian Fish. Sci. 3(2):185-196.

Liao, C.-I., H.-M. Su and E.Y. Chang, 2001 Techniques in finfish larviculture in Taiwan. Aquaculture 200(2001):1-31.

Manea, G., 1985 Acclimatization of new fishes and other aquatic organisms. Ceres Publishing House, Bucharest.

Manna, G.K. and A.R. Khuda-Bukhsh, 1977 Karyomorphology of cyprinid fishes and cytological evaluation of the family. Nucleus. 20:119-127.

Courtenay, Jr. and J.R. Stauffer (eds.) Distribution, biology and management of exotic fishes. Johns Hopkins University Press, Baltimore, USA.

Nichols, J.T., 1943 The freshwater fishes of China. Natural history of Central Asia: Volume IX. The American Museum of Natural History, New York, USA, 322 p.

Panov, D.A., Y.I. Sorokin and L.G. Montenkova, 1969 Experimental study of the feeding of young silver carp (Hypophthalmichthys molitrix). Prob. Ichthyol. 9:101-112.

Pantastico, J.B., S.F. Baldia and J.P. Baldia, 1986 Efficiency of some cyanophytes as larval feed for silver carp (Hypophthalmichthys molitrix) and the culture of Spirulina platensis. p. 609-614. In J.L. Maclean, L.B. Dizon and L.V. Hosillos (eds.) The First Asian Fisheries Forum. Asian Fisheries Society, Manila, Philippines. p. 609-614

Paulin, C., A. Stewart, C. Roberts and P. McMillan, 1989 New Zealand fish: a complete guide. National Museum of New Zealand Miscellaneous Series No. 19. 279 p.

Reddy, P.V.G.K., 1991 A comparative study of the karyomorphology of grass carp, Ctenopharyngodon idella (Val.), silver carp, Hypophthalmichthys molitrix (Val.) and their F1 hybrid. J. Aquacult. 1:31-41

Ren, X., J. Cui, and Q. Yu, 1992 Chromosomal heterochromatin differentiation of cyprinid fishes. Genetica 87:47-51

Robins, C.R., R.M. Bailey, C.E. Bond, J.R. Brooker, E.A. Lachner, R.N. Lea and W.B. Scott, 1991 Common and scientific names of fishes from the United States and Canada. Am. Fish. Soc. Spec. Pub. (20):183 p.

Robins, C.R., R.M. Bailey, C.E. Bond, J.R. Brooker, E.A. Lachner, R.N. Lea and W.B. Scott, 1991 World fishes important to North Americans. Exclusive of species from the continental waters of the United States and Canada. Am. Fish. Soc. Spec. Publ. (21):243 p.

Schoonbee, H.J., F. De W. Brandt and C.A.L. Bekker, 1978 Induced spawning of the two phytophagous Chine carp species Ctenopharyngodon idella (Val.) and Hypophthalmychthys molitrix (Val.) with species reference to possible use of grass carp in the control of aquatic weeds. Water S.A. 4(2):93-103.

Welcomme, R.L., 1988 International introductions of inland aquatic species. FAO Fish. Tech. Pap. 294. 318 p. 128-132

Zan, R. and Z. Song, 1980 Comparative analysis of karyotypes of Cyprinus carpio and Carassius auratus, as well Aristichthys nobilis and Hypophthalmichthys molitrix. Acta genet. sin. 7(1):72-76.

Zan, R., Z. Song, and W. Liu, 1986 Studies on karyotypes and DNA contents of some cyprinoid fishes, with notes on fish polyploids in China. In: T. Uyeno, R. Arai, T. Taniuchi, and K. Matsuura (eds), Indo Pacific Fish Biology, Tokyo, Ichthyol. Soc. of Japan, p.877-885.

[11]***Mylopharyngodon piceus*** (Richardson 1846)

Common Name: black carp

Taxonomy and identification. A few distinguishing characteristics were provided by Berg (1949), Masuda et al. (1984), and Howells (1992b). Black carp superficially resemble grass carp (*Ctenopharyngodon idella*), but there are no known voucher specimens in US museums.

Size: maximum size recorded 122 to 131 cm TL and 32 to 36 kg (males/unsexed individuals)

Life history and general biology. Black carp are demersal freshwater fish, ranging from near surface to a depth of 10m. As a bottom-dwelling molluscivore, black carp were imported into the United States in the early 1970's for use as a food fish and also as a biological control agent for snails that serve as an intermediate host for a trematode parasite in fish held captive on fish farms. More recently, this species has been proposed as a biological control for the introduced zebra mussel *Dreissena polymorpha*.

While adults preferentially prey on mollusk, black carp diet varies with age. Immatures forage on zooplankton and fingerling fishes, and adults will prey on benthic crustaceans, aquatic insects, and fish eggs when mollusks are not available. As immatures and adults, black carp, if established in the wild, could have serious adverse impacts, due to predation on native mollusc species (including threatened and endangered species) and on fingernail clams which are a primary food source of migrating waterfowl and fish. For example, black carp commonly feed on mollusks, using their pharyngeal teeth to crush the mollusks shells, then consuming the soft tissues, e.g., a 4-year old juveniles are capable of consuming ca. 1-2 kg of molluscs per day. Given their preference of mollusks, black carp were considered at biological control agents for zebra mussel (e.g., French 1993; Rubinshtein 1994; Ricciardi 1994), but there is little experimental evidence that indicates black carp would be effective in controlling zebra mussels. Because black carp do not have jaw teeth and their mouths are relatively small, it is unlikely that these fish are capable of breaking apart zebra mussel rafts (Nico and Williams 1996).

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[11]Original material posted to FishBase, Leo Nico and Pam Fuller (Center for Aquatic Resource Studies, USGS/BRD, Danielle M. Crosier and Daniel P. Molloy (New York State Museum) with assistance from Jerry Rasmussen (U.S. Fish and Wildlife Service). Last accessed November, 2004. Updated and edited, June, 2004 through November, 2004.

Mylopharyngodon piceus characteristically presents black-tipped scales that appear as cross-hatching upon viewing. Diagnostically, the dorsal fin is located above the pelvic fins, and is short and pointed, containing 7-8 rays. The anal fin is located closer to the caudal fin than in the native minnow. While the black carp resembles the grass carp in size, color, and appearance (e.g., shape of fins, and size and position of eyes), the species may be distinguished on the basis of their pharyngeal teeth (i.e., pharyngeal teeth of the grass carp possess deep parallel grooves and those of black carp appear molar-like).

Black carp are superficially very similar in appearance to grass carp, *Ctenopharyngodon idella*, specifically in terms of body size and shape, position and size of fins, and position and size of the eyes. Juveniles, in particular, are difficult to distinguish from grass carp young. As such, Nico and Williams (1996) expressed concern that if black carp become more common in U.S. aquaculture, there will be an increased risk that the species be misidentified and unintentionally introduced as "grass carp" to some areas. Available information indicates that the black carp is, or has been in the recent past, maintained in research or production facilities in six states including Arkansas, Louisiana, Mississippi, Missouri, North Carolina, Oklahoma, and Texas (Nico and Williams 1996). There is widespread concern, however, that black carp will escape captivity, establish wild populations, and cause major adverse environmental impacts. In addition, black carp are host to parasites, flukes, and bacterial and viral diseases and could possibly transfer these to other fish species.

The reproductive biology of the black carp is similar to other members of the family. Females are capable of producing between 129,000 and 1,180,000 eggs per year (depending upon body size), and are deposited on the bottom in a single batch. Fertilization occurs upstream and eggs drift downstream with the current until reaching areas with little current (e.g., floodplain lakes, smaller streams, and water channels). Following fertilization, the bathypelagic eggs become hydrated and swell 4- to 5-fold, and are carried by currents. Embryos typically pass through 8 embryonic stages, followed by 4 larval stages, and then two stages as fry. Early developmental stages are particularly sensitive to infections (bacterial and fungal), and growth rate before sexual maturation is determined especially by quality and quantity of food. Maturity is reached at 6 to 11 years of age in the species native habitat, and in captivity maximum lifespan is 15 years. Growth continues after sexual maturity is reached, and relatively large increments in length and weight occur annually. Once maturity has been reached, black carp are capable of annual reproductive activity. Population doubling times are relative low, with a minimum ranging between 4.5 and 14 years. While temperature preferences and lower temperature tolerances are poorly characterized, in the species native range spawning begins when food is available, water levels are high, and water temperatures reach 26-30ºC.

Native Range. Their native distribution is primarily subtropical, lying between 53ºN to 15ºN where the species occupies freshwater habitats in most major Pacific drainages of eastern Asia

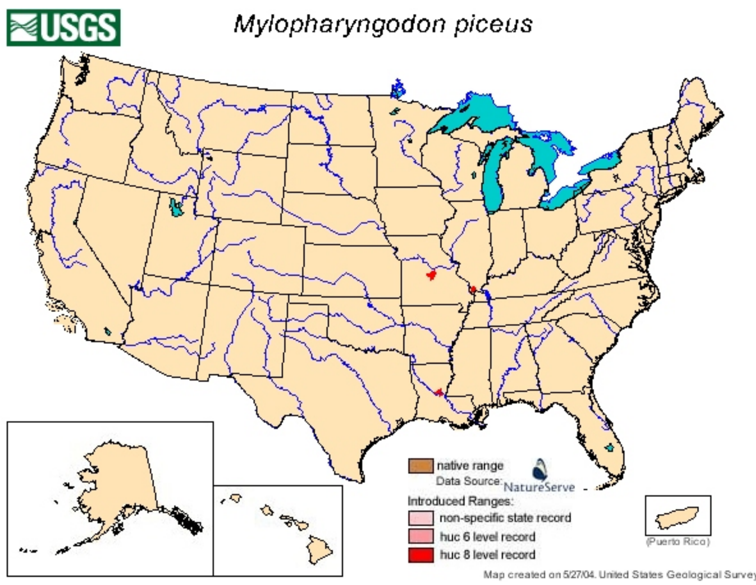

Figure 14. Known distribution of black carp in US.

from the Pearl River (Zhu Jiang) basin in the north of China to the Amur River (Heilong Jiang) and its major tributaries of China and far eastern Russia. Black carp may have been native to the Honghe or Red River of northern Vietnam (Nico and Williams 1996).

Nonindigenous occurrences and current status. Introductions of *Mylopharyngodon piceus* documented in FishBase (n = 31) clearly indicate that invasions of the species have occurred worldwide, although not all invasions develop sustainable populations subsequent to their release. From its native distribution in China, *Mylopharyngodon piceus* has expanded its disjunct distribution throughout Asia during the 19th and early 20th century. In the 1980's the species invaded the Nearctic region with documented releases occurring in the Carribbean, although establishment of sustainable populations are few, if any. In the US (Figure 14), black carp have been maintained in hatcheries, fish culture facilities, and fish farm ponds (mainly located in the southeastern United States). Both diploid (fertile) and triploid (sterile) fish have been used in fish farming applications, and some have escaped captivity. Approximately 30 black carp escaped from a fish farm in Missouri into the Osage River, Missouri River basin, during a flood event in

April 1994 (Anonymous 1994b; W. Pflieger, personal communication). The first specimen reported from the wild was captured by a commercial fisherman in March 2003 from Horseshoe Lake in Illinois near the confluence of the Mississippi and Ohio Rivers. Preliminary analyses indicate that this fish (4-year old; 783 mm, 5.8 kg) was a triploid individual. A second specimen was captured from the wild in the lower Red River, Louisiana in April 2004.

Means of Introduction. This species was first brought into the United States in the early 1970's with imported grass carp stocks. These fish came from Asia and were sent to a private fish farm in Arkansas (Nico and Williams 1996). The second introduction of black carp into this country occurred in the early 1980's. During this period it was imported as a a food fish and as a biological control agent to combat the spread of yellow grub *Clinostomum margaritum* in aquaculture ponds (Nico and Williams 1996). Current reports indicate that black carp are established in Illinois and Missouri, but it is not clear if the Illinois specimen resulted from the Missouri escape. As previously noted, the black carp captured from the wild may have escaped in Missouri and was thought to be triploid (Anonymous 1994b). However, it also was rumored that these fish may have been brood stock, and the commercial fisherman who captured the Louisiana specimen reported catching a “different-looking grass carp” for the past eight years. Capture of black carp in Louisiana probably stem from escape from an aquaculture facility.

Impact of introduction and control measures. Black carp are considered a potential pest species, particularly given its similarities to grass carp (*Ctenopharyngodon idella*) with respect to the species’ reproductive biology. Given that the grass carp has expanded its range since its introduction to the US in 1963, a current concern among those working with invasive species is, black carp might be capable of expanding their distribution in a similar manner to grass carp, if the opportunity were presented. If so, there is high potential that the black carp would negatively impact native aquatic communities by feeding on, and reducing, populations of native mussels and snails, many of which are considered endangered or threatened (Nico and Williams 1996). At present, steps have been taken to regulate the culture of black carp by placing it on the federal list of injurious wildlife species under the Lacey Act, which may help contain managed populations. Trade of the species is restricted in Germany, and several countries report adverse ecological impact after introduction of black carp.

References with Supplemental Bibliography (derived from FishBase)

Anon., 1994 Atlas pesquero de México. Instituto Nacional de la Pesca. 234 p.

Anon., 2002, Fish collection database of the American Museum of Natural History American Museum of Natural History, Central Park West at 79th Street, New York, NY 10024-5192, USA

Anon., 2001Fish collection database of the National Museum of Natural History (Smithsonian Institution). Smithsonian Institution - Division of Fishes.

Cahoon, W. G. (1953). Commercial carp removal at Lake Mattamuskeet, North Carolina, *Journal of Wildlife Management* 17(3):312-317.

Carp Task Force. (1989). *Report to the Louisiana Legislature, March 1989*. Three volumes, Louisiana Department of Wildlife and Fisheries, Baton Rouge, LA.

Chick, J.H., R.J. Maher, B.M. Burr and M.R. Thomas, 2003 First black carp captured in U.S. Science 300:1876-1877.

Collins, C. (1996). The Chinese black carp: A potential biological control for snails in warmwater fish production ponds, *Aquaculture Magazine* 22(3):83-86.

Cui, J., X, Ren and Yu, Q., 1991 Nuclear DNA content variation in fishes. Cytologia 56:425-429.

Eschmeyer, W.N., Editor, 1999 Catalog of fishes. Updated database version of November 1999. Catalog databases as made available to FishBase in November 1999.

Evtushenko, N. Y., Potrokhov, A. S., and Zin'kovskii, O. G. (1994). The black carp as a subject for acclimatization (Review), *Hydrobiological Journal* 30(5):1-10.

Garibaldi, L., 1996 List of animal species used in aquaculture. FAO Fish. Circ. 914. 38 p.

Holcík, J., 1991, Fish introductions in Europe with particular reference to its central and eastern part. Can. J. Fish. Aquat. Sci. 48 (Suppl. 1):13-23.

Klinkhardt, M, M. Tesche and H. Greven, 1995 Database of fish chromosomes. Westarp Wissenschaften.

Kottelat, M., 1997 European freshwater fishes. Biologia 52, Suppl. 5:1-271.

Lou, Y.L., K.Y. Zhang, Y.L. Wu and Y.M. Wang., 1983 Studies on karyotypes of black carp (Mylopharyngodon piceus). J. Fish China 7(1):77-81.

Marsden, J. E. (1997). Common carp diet includes zebra mussels and lake trout eggs, *Journal of Freshwater Ecology* 12(3):491-492.

Nichols, J.T., 1943The freshwater fishes of China. Natural history of Central Asia: Volume IX. The American Museum of Natural History, New York, USA, 322 p.

Nico, L. G., and Williams, J. D. (1996). Risk assessment on black carp (Pisces: Cyprinidae), Final Report to the Risk Assessment and Management Committee of the Aquatic Nuisance Species Task Force. U.S. Geological Survey, Biological Resources Division, Gainesville, FL.

Robins, C.R., R.M. Bailey, C.E. Bond, J.R. Brooker, E.A. Lachner, R.N. Lea and W.B. Scott, 1991World fishes important to North Americans. Exclusive of species from the continental waters of the United States and Canada. Am. Fish. Soc. Spec. Publ. (21):243 p.

Shields, J. T. (1958). Experimental control of carp reproduction through water drawdowns in Fort Randall Reservoir, South Dakota. *Transactions of the American Fisheries Society.* 87(1957):23-33.

Smiley, C. W. (1886). Some results of carp culture in the United States, Report of the Commissioner of Fish and Fisheries for 1884, Part XII, U.S. Commission of Fish and Fisheries, Washington, DC, 657-890.

Welcomme, R.L., 1988 International introductions of inland aquatic species. FAO Fish. Tech. Pap. No. 294. 318 p.

[12]***Ctenopharyngodon idella*** (Valenciennes 1844)

Common Name: grass carp, white amur

Size: Maximum size ranges between 125 to 150 cm TL (male/unsexed; Ref. 30578) with a maximum recorded weight of 45.0 kg (Ref. 7248)

Taxonomy and identification. Distinguishing characteristics for grass carp were given in Berg (1949), Shireman and Smith (1983), and Page and Burr (1991), and more recently published state and regional fish books provide life histories and photographs of the species (e.g., Robison and Buchanan 1988; Etnier and Starnes 1993; Jenkins and Burkhead 1994; Pflieger 1997). A few authors identify the scientific name as *Ctenopharyngodon idellus*, but that term is not valid.

Diagnotically, the species is well characterized, having dorsal spines (total): 3-3; dorsal soft rays (total): 7-8; anal spines: 3-3; and anal soft rays: 7-11. Barbels are absent from a very shout snout (length less than or equal to eye diameter) and the postorbital length is more than half the length of the head. The caudal fin has 18 soft rays.

Life history and biology. Grass carp occur in a wide range of aquatic habitats, ranging from those presenting relatively fast currents to lakes, ponds, pools and backwaters of large rivers where the species prefers large, slow-flowing or standing water bodies with vegetation. The species tends toward being demersal and usually ranges from near surface to a depth of 5 m. The native range of grass carp was temperate, mid-latitude (65˚N - 25˚N), with a normal temperature range from 10 - 26˚C. However, the species tolerants wide range of environmental conditions, which in part supports their invasiveness attributes. For example, grass carp are relatively tolerant to temperatures from 0º to 38ºC, salinities as greater as 10 ppt, and dissolved oxygen levels as low as 0.5 ppm. Grass carp feed on aquatic vascular plants, detritus, insects and other invertebrates. The species has a long history as an aquaculture species commonly used for weed control in rivers, fish ponds and reservoirs.

Reproduction biology of grass carp is typical of “Asian carp” with spawning generally initiated

[12]Original material prepared by Leo Nico and Pam Fuller (January 4, 2001) at USGS/BRD/FISC. Updated, expanded with FishBase output, and edited June, 2004 through November, 2004.

along riverbeds with relatively strong current. Embryo-larval development is similiar to other carp, with a maximum age recorded at 21 years. While used extensively throughout the world for aquatic vegetation control, the species is also used as food for human consumption. The species is widely regarded as a pest in most countries because of the damages made to submerged vegetation when the species escapes from its intended role control in nuisance aquatic vegetation.

Native Range. As with other "Asian carp," the original distribution of grass carp included eastern Asia from the Amur River of eastern Russia and China south to West River of southern China (Lee et al. 1980. Shireman and Smith 1983), China and Eastern Siberia (i.e., Amur River system) and widely transported around the world.

Nonindigenous Occurrences. Currently, in the US the grass carp occupies a wide, disjunct distribution (Figure 15) which in part reflects the species opportunistic dispersal associated with intentional releases. The distribution depicted in Figure 16 results from extrapolation of point-sourced releases documented in the literature and summarized in the dot map in Figure 15.

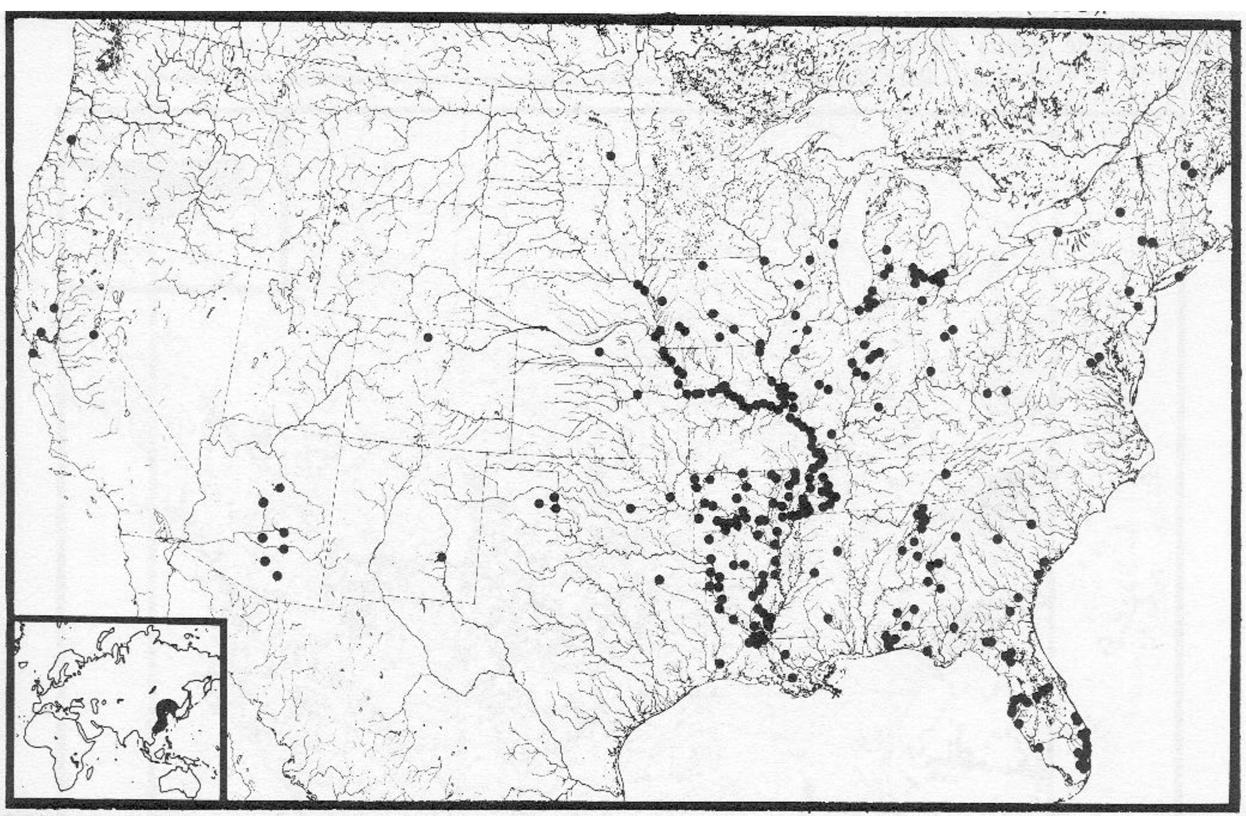

Figure 15. Dot map of grass carp in surface waters of the US.

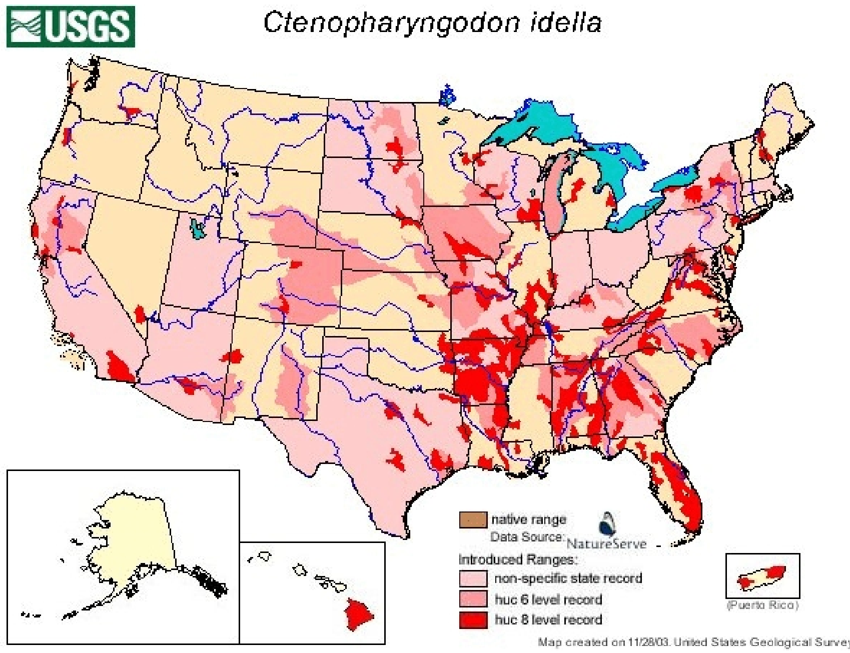

Figure 16. Projected current distributions for grass carp developed form dot map, e.g., in Figure 15.

Grass carp have been recorded from Alabama (Guillory and Gasaway 1978; Boschung 1992; Kirk et al. 1994; Mettee et al. 1996); Arizona (Minckley 1973; Guillory and Gasaway 1978; Courtenay et al. 1984, 1991); Arkansas (Buchanan 1973; Guillory and Gasaway 1978; Zimpfer et al. 1987); California (Guillory and Gasaway 1978; Courtenay et al. 1984, 1991; Dill and Cordone 1997); Colorado (Guillory and Gasaway 1978; Courtenay et al. 1984, 1991; Woodling 1985); Connecticut (Whitworth 1996); Delaware (Courtenay et al. 1984, 1991; Raasch and Altemus 1991; Rohde et al. 1994); Florida (Guillory and Gasaway 1978; Courtenay and Stauffer 1984; Florida Game and Freshwater Fish Commission 1989, 1994; Shafland 1995b); Georgia (Guillory and Gasaway 1978; Courtenay et al. 1984; Walters 1997); Hawaii (Maciolek 1984); Idaho (Courtenay et al. 1984, 1991; Idaho Fish and Game 1990); Illinois (Pflieger 1975; Anonymous 1977; Guillory and Gasaway 1978; Smith 1979; Phillips et al. 1982; Burr and Page 1986; Burr et al. 1996; Laird and Page 1996); Indiana (Anonymous 1977; Guillory and Gasaway 1978; Simon et al. 1992); Iowa (Guillory and Gasaway 1978; Burr and Page 1986; Harlan et al. 1987; Courtenay et al. 1991); Kansas (Guillory and Gasaway 1978; Courtenay and Williams 1992; Cross and Collins 1995); Kentucky (Conner et al. 1980; Courtenay et al. 1984, 1991; Burr and Page 1986; Burr and Warren 1986); Louisiana (Guillory and Gasaway 1978; Conner and Suttkus 1986; Zimpfer et al. 1987; Carp Task Force 1989); Maryland (Guillory and Gasaway 1978; Courtenay et al. 1984, 1991; Rohde et al. 1994); Massachusetts (Courtenay et al. 1984, 1991; Hartel 1992; Hartel et al. 1996); Michigan (Guillory and Gasaway 1978; Lee et al. 1980 et seq.;

Courtenay et al. 1984; Emery 1985); Minnesota (Phillips et al. 1982; Courtenay et al. 1984, 1991); Mississippi (Guillory and Gasaway 1978; Courtenay et al. 1991; Courtenay 1993); Missouri (Pflieger 1975, 1978, 1997; Guillory and Gasaway 1978; Brown and Coon 1991); Nebraska (Guillory and Gasaway 1978; Courtenay et al. 1984, 1991); Nevada (Courtenay et al. 1984, 1991; Deacon and Williams 1984); New Hampshire (Guillory and Gasaway 1978; Lee et al. 1980 et seq.; Schmidt 1986); New Jersey (Guillory and Gasaway 1978; D. Mitchell and Soldwedel, personal communication); New Mexico (Guillory and Gasaway 1978; Courtenay et al. 1984, 1991; Cowley and Sublette 1987; Sublette et al. 1990); New York (Guillory and Gasaway 1978; Courtenay et al. 1984, 1991; Smith 1985; Schmidt 1986); North Carolina (Guillory and Gasaway 1978; Courtenay et al. 1984, 1991; Menhinick 1991; Rohde et al. 1994); North Dakota (Lee et al. 1980 et seq.; Owen et al. 1981; Power and Ryckman 1998); Ohio (Guillory and Gasaway 1978; Courtenay et al. 1984, 1991); Oklahoma (Guillory and Gasaway 1978; Courtenay et al. 1984, Cashner and Matthews 1988; Pigg et al. 1992); Oregon (Lee et al. 1980 et seq.; Pauley et al. 1994); Pennsylvania (C. N. Shiffer, personal communication); South Carolina (Guillory and Gasaway 1978; Courtenay et al. 1984, 1991; Foltz and Kirk 1994; Rohde et al. 1994); South Dakota (Guillory and Gasaway 1978; Lee et al. 1980 et seq.; Owen et al. 1981); Tennessee (Guillory and Gasaway 1978; Ryon and Loar 1988; Etnier and Starnes 1993); Texas (Guillory and Gasaway 1978; Conner and Suttkus 1986; Trimm et al. 1989; Howells 1992a); Utah (Courtenay et al. 1984, 1991; Sigler and Sigler 1996); Virginia (Guillory and Gasaway 1978; Courtenay et al. 1984, 1991; Jenkins and Burkhead 1994; Rohde et al. 1994); Washington (Pauley et al. 1994; Fletcher, personal communication); West Virginia (Guillory and Gasaway 1978; Courtenay et al. 1991); Wisconsin (Guillory and Gasaway 1978; Becker 1983; Emery 1985; Burr and Page 1986; Mulvey 1990; Fago 1992); and Wyoming (Courtenay et al. 1984, 1991; Stone 1995).

Means of introduction and status. Both authorized and unauthorized stockings of grass carp have taken place for biological control of vegetation. This species was first imported to the United States in 1963 to aquaculture facilities in Auburn, Alabama, and Stuttgart, Arkansas. The Auburn stock came from Taiwan, and the Arkansas stock was imported from Malaysia (Courtenay et al. 1984).

The first release of this species into open waters took place at Stuttgart, Arkansas, when fish escaped the Fish Farming Experimental Station (Courtenay et al. 1984). However, many of the early stockings in Arkansas were in lakes or reservoirs open to stream systems, and by the early 1970s there were many reports of grass carp captured in the Missouri and Mississippi rivers

(Pflieger 1975, 1997). During the past few decades, the species has spread rapidly as a result of widely scattered research projects, stockings by federal, state, and local government agencies, legal and illegal interstate transport and release by individuals and private groups, escapes from farm ponds and aquaculture facilities; and natural dispersal from introduction sites (e.g., Pflieger 1975; Lee et al. 1980 et seq.; Dill and Cordone 1997). Some of the agencies that have stocked grass carp in the past include the Arkansas Game and Fish Commission, the Tennessee Valley Authority, the U.S. Fish and Wildlife Service, the Delaware Division of Fish and Wildlife, the Florida Game and Fresh Water Fish Commission, the Iowa Conservation Commission, the New Mexico Department of Fish and Game, and the Texas Parks and Wildlife Department. The species also has been stocked by private individuals and organizations. In some cases, grass carp have escaped from stocked waterbodies and appeared in nearby waterbodies. Stocking of grass carp as a biological control against nuisance aquatic plants in ponds and lakes continues. For instance, Pflieger (1997) stated that thousands of grass carp are reared and sold by fish farmers in Missouri and Arkansas.

In total, grass carp are presently reported from 45 states; there are no reports of introductions in Alaska, Maine, Montana, Rhode Island, and Vermont. It is known to have established populations in a number of states in the Mississippi River basin. Breeding populations have been recorded for the Mississippi River in Kentucky (Conner et al. 1980; Burr and Warren 1986), the Illinois and upper Mississippi rivers of Illinois and Missouri (Raibley et al. 1995), the lower Missouri River in Missouri (Raibley et al. 1995), the Mississippi River or its tributaries in the states of Arkansas (Conner et al. 1980), Louisiana (Conner et al. 1980; Zimpfer et al. 1987), Tennessee (Etnier and Starnes 1993), and presumably Mississippi (Courtenay et al. 1991). It is also established in the Ohio River in Illinois (Burr, personal communication); it was listed as established in Minnesota (Courtenay et al. 1991, but see Courtenay 1993), and in the Trinity River of Texas (Waldrip 1992; Webb et al. 1994; Elder and Murphy 1997). Courtenay (1993) listed grass carp as established in eight states, Arkansas, Kentucky, Illinois, Louisiana, Missouri, Mississippi, Tennessee, and Texas; an additional one, Minnesota, was included in an earlier listing of states with established populations (Courtenay et al. 1991). Stone (1995) listed this species as being established in Wyoming; however, Stone (personal communication) clarified his earlier report by stating that, as of early 1997, there is no evidence of natural reproduction in that state. Similar to a few other authors, he used the term “established” to indicate that grass carp populations have persisted for many years, presumably because of their long life span and because of long-term maintenance of wild populations through continued stockings. Pearson and Krumholz (1984) mentioned several records from the Ohio River, including river mile 963 on the Illinois-Kentucky border and from the Falls of the Ohio, at Louisville, along the Kentucky-Ohio border. They also stated that the species had been stocked in many private ponds and lakes in the Ohio River basin. Sigler and Sigler (1996) stated that this species is no longer found in Utah, but they provide no details. Harvest of grass carp by commercial fishermen in the Missouri and Mississippi rivers of Missouri has exhibited a general climb. In 1996, the most recent available data, there was a record reported harvest, about 44,000 pounds, 8 percent of the total commercial fish harvest (J. W. Robinson, personal communication).

Impact of introduction. Outside the species' native range in China and Eastern Siberia (Amur River system), grass carp have been widely transported around the world, and several countries report adverse ecological impacts after introduction of grass carp. Various authors (e.g., Shireman and Smith 1983; Chilton and Muoneke 1992; Bain 1993) have reviewed the literature on grass carp, and most discuss actual and potential impacts caused by the species' introduction.

Shireman and Smith (1983) concluded that the effects of grass carp introduction on a water body are complex and apparently depend on the stocking rate, macrophyte abundance, and community structure of the ecosystem. The authors indicated that numerous contradictory results are reported in the literature concerning grass carp interaction with other species. Negative effects involving grass carp reported in the literature and summarized by these authors included interspecific competition for food with invertebrates (e.g., crayfish) and other fishes, significant changes in the composition of macrophyte, phytoplankton, and invertebrate communities, interference with the reproduction of other fishes, and decreases in refugia for other fishes.

In their overview, Chilton and Muoneke (1992) reported that grass carp seem to indirectly affect other animal species by modifying preferred habitat. However, the authors also indicated that grass carp may directly influence other animals through either predation or competition when plant food is scarce. Similarly, Bain (1993) stated that grass carp have significantly altered the food web and trophic structure of aquatic systems by inducing changes in plant, invertebrate, and fish communities. Bain (1993) indicated that effects are largely secondary consequences of decreases in the density and composition of aquatic plant communities. Organisms requiring limnetic habitats and food webs based on phytoplankton tend to benefit from the presence of grass carp. On the other hand, Bain (1993) also reported that declines have occurred in the diversity and density of organisms that require structured littoral habitats and food chains based on plant detritus, macrophytes, and attached algae.

Removal of vegetation can have negative effects on native fish, such as elimination of food sources, shelter, and spawning substrates (Taylor et al. 1984). Hubert (1994) similarly cited a study that found vegetation removal by grass carp lead to better growth of rainbow trout due to increases in phytoplankton and zooplankton production, but it also lead to higher predation on rainbow trout by cormorants *Phalacrocorax auritus* due to lack of cover, and changes in diet, densities, and growth of native fishes. Although grass carp are often used to control selected aquatic weeds, these fish sometimes feed on preferred rather than on target plant species (Taylor et al. 1984). Increases in phytoplankton populations is a secondary effect of grass carp presence. A single grass carp can digest only about half of the approximately 45 kg of plant material that it consumes each day. Collateral to these grazing effects, the deposition of undigested material and feces into aquatic habitats by grass carp promotes nutrient enrichment which often promotes algal blooms (Rose 1972). These blooms can subsequently reduce water clarity and decrease oxygen levels (Bain 1993).

In addition to problems related to switching of food sources, grass carp may also carry several parasites and diseases known to be transmissible or potentially transmissible to native fishes. For example, grass carp imported from China may have been the source of introduction of the Asian tapeworm *Bothriocephalus opsarichthydis* (Hoffman and Schubert 1984; Ganzhorn et al. 1992), and the species may have been indirectly responsible for infection of the endangered woundfin *Plagopterus argentissimus* (by way of the red shiner *Cyprinella lutrensis*; Moyle 1993). If parasites of the grass carp can jump to native cyprinidae, introductions of grass carp may have also present opportunity for other diseases of *Ctenopharyngodon idella* (see Table 1) to expand their range.

Although triploids are considered sterile and incapable of reproduction, some researchers have questioned the sterility of triploids. Howells (1992b) referred to a study in which milt from triploids was used to successfully fertilize normal diploid eggs, but he found no published information on the viability of eggs from triploid females. Available information (e.g., Chilton and Muoneke 1992; Exotic Species Workgroup 1994; W. Shelton, personal communication) indicate that triploid grass carp can produce some viable gametes, but the proportion of such gametes is extremely low; hence, the reproductive potential of triploids is considered to be very low (Chilton and Muoneke 1992). In general, triploid females never fully develop ovaries, but triploid males may present a superfiscal appearance of being fertile with a complement of fully developed testes in the adult. Techniques to induce triploidy are not always totally effective and every individual needs to be genetically checked. Triploid grass carp are indistinguishable in external morphology from normal (fertile) diploids, so triploidy must be confirmed by blood or tissue analysis.

Case history suggests that grass carp management must be practiced cautiously. For example, although diploid grass carp are banned in Florida, in 1991 three adults, including a gravid female (UF 85587), were captured in the Suwannee River, Florida (Burkhead and Williams 1991). Similarly, Webb et al. (1994) reported on the capture of young grass carp in Texas whose ploidy and age indicated some were escapees from illegal stockings. Although triploids are generally allowed, many states no longer permit import of diploid grass carp. Some states, however, are exercising additional precautions, e.g., Texas restricts grass carp releases, given the species' history of excessive removal of vegetation and the subsequent destruction of fish and wildlife habitat. As of 1994, Alaska, Oregon, Montana, North Dakota, Minnesota, Wisconsin, Michigan, Massachusetts, Vermont, Maine, Maryland, and Rhode Island also prohibit grass carp, diploid and triploid, in their state. The states of Hawaii, Iowa, Kansas, Missouri, Oklahoma, Arkansas, Mississippi, and Alabama have no restrictions, meaning that both diploids and triploids can be used and there is no permit required. The remaining states have some type of restriction on the use of grass carp. Typical restrictions include one or more of the following: use of only verified triploids, use only in public waters, or use requires a permit (Wattendorf and Phillippy 1996).

The numbers of grass carp legally stocked in a region can be fairly high. For instance, in January 1992, the Texas Parks and Wildlife Commission approved the use of certified triploid grass carp for vegetation control in state waters. In March 1994, Durocher (1994) reported that the relatively new program already had issued nearly 2,900 permits and allowed the stocking of more than 78,000 grass carp. Foltz and Kirk (1994) indicated that 300,000 triploid grass carp were stocked into Lake Marion, South Carolina, between 1989 and 1991. Since the late 1970's more than 2,000 water bodies in Florida have been stocked with grass carp (Haller 1994). Lake Istokpoga, Florida, was stocked with 125,000 triploid grass carp during a two-year period in the early 1990s (Thomas 1994). Guillory and Gasaway (1978) gave dot map detailing the known distribution of grass carp at that time. Pflieger (1978) described the invasion of Missouri streams. Dill and Cordone (1997) detailed the history of grass carp introductions in California. As part of their account, the authors reported the removal of about 1,500 diploid grass carp from golf course ponds that had been illegally stocked during the 1980's.

References and Supplemental Bibliography (derived from FishBase)

Roberts, R.J. (ed.), 1978 Fish pathology. Cassell Ltd., London. 318 p.

van Duijn, C. Jr., 1973 Diseases of fishes. Illffe Books Ltd., London. 372 p.

Utter, F. and L. Folmar, 1978Protein systems of grass carp: allelic variants and their application to management of introduced populations. Trans. Am. Fish. Soc. 107(1):129-134.

Opuszynski, K., 1972 Use of phytophagous fish to control aquatic plants. Aquaculture 1(1):61-74.

Sinha, V.R.P. and M.V. Gupta, 1975 On the growth of grass carp, Ctenopharyngodon idella Val. in composite fish culture at Kalyani, West Bengal. Aquaculture 5:283-290.

Gorbach, E.I., 1972 Fecundity of the grass carp (Ctenopharyngodon idella Val.) in the Amur basin. J. Ichthyol. 12(4):616-625.

Kilambi, R.V. and A. Zdinak, 1981 Comparison of early development stages and adults of grass carp, Ctenopharyngodon idella, and hybrid carp (female grass carp x male bighead Aristichthys nobilis). J. Fish Biol. 19:459-465.

Woynarovich, E. and L. Horvath, 1980 The artificial propagation of warm-water finfishes - a manual for extension. FAO Fish. Tech. Pap. (201):183 p.

Yu, Y.S., A.W. Sin and Y.W. Chan, 1973 Results of induced spawning of the bighead, Aristichthys nobilis (Richardson), grass carp, Ctenopharyngodon idella (Valenciennes) and silver carp, Hypophthalmichthys molitrix (Valenciennes) in Hong Kong, 1969-71. Hong Kong Fish. Bull. (3):55-76.

Robins, C.R., R.M. Bailey, C.E. Bond, J.R. Brooker, E.A. Lachner, R.N. Lea and W.B. Scott, 1980 A list of common and scientific names of fishes from the United States and Canada. Am. Fish. Soc. Spec. Publ. (12)1-174.

Welcomme, R.L., 1988 International introductions of inland aquatic species. FAO Fish. Tech. Pap. No. 294. 318 p.

Reimans, R.G., 1978 Grass carp as a tool in fishery management. Proc. Eur. Weed Res. Symp. Aquat. Weeds (5).

Mitchell, C., 1977 The use of grass carp for submerged weed control. Proc. N.Z. Weed Pests Control Conf. 30:145-148.

Robins, C.R., R.M. Bailey, C.E. Bond, J.R. Brooker, E.A. Lachner, R.N. Lea and W.B. Scott, 1991 Common and scientific names of fishes from the United States and Canada. Am. Fish. Soc. Spec. Pub. (20):183 p.

Shireman, J.V., 1975 Predation, spawning and culture of white amur (Ctenopharyngodon idella). Ann. Rep. Fla. Dept. Nat. Res., Univ. Fla., School of Forest Resources and Conservation, Gainesville, Florida. 40 p.

Yamamoto, M.N., 1992, Occurrence, distribution and abundance of accidentally introduced freshwater aquatic organisms in Hawaii. State of Hawaii, Federal Aid in Sportfish Restoration, Dingell-Johnson JOR. Freshwater Fisheries Research and Surveys, Project No. F-14-R-16.

Page, L.M. and B.M. Burr, 1991 A field guide to freshwater fishes of North America north of Mexico. Houghton Mifflin Company, Boston. 432 p.

Courtenay, W.R. Jr., D.A. Hensley, J.N. Taylor and J.A. McCann, 1984, Distribution of exotic fishes in the continental United States. p. 41-77. In W.R. Courtenay, Jr. and J.R. Stauffer, Jr. (eds.) Distribution, biology and management of exotic fishes. Johns Hopkins University Press, Baltimore, USA.

Crossman, E.J., 1991, Introduced freshwater fishes: A review of the North American perspective with emphasis on Canada. Can. J. Fish. Aquat. Sci. 48 (Suppl. 1):46-57.

Erdman, D.S., 1984, Exotic fishes in Puerto Rico. p. 162-176. In W.R. Courtney, Jr. and J.R. Stauffer, Jr. (eds.) Distribution, biology and management of exotic fishes. Johns Hopkins University Press, Baltimore, USA.

Contreras-B, S. and M.A. Escalante-C, 1984, Distribution and known impacts of exotic fishes in Mexico. p. 102-130. In W.R. Courtenay, Jr. and J.R. Stauffer, Jr. (eds.) Distribution, biology and management of exotic fishes. Johns Hopkins University Press, Baltimore, USA.

Anon., 1996 Fish collection database of the University of British Columbia Fish Museum Fish

Museum University of British Columbia, Vancouver, Canada.

Garibaldi, L., 1996 List of animal species used in aquaculture. FAO Fish. Circ. 914. 38 p.

Hugg, D.O., 1996, MAPFISH georeferenced mapping database. Freshwater and estuarine fishes of North America. Life Science Software. Dennis O. and Steven Hugg, 1278 Turkey Point Road, Edgewater, Maryland, USA.

Lever, C., 1996 Naturalized fishes of the world. Academic Press, California, USA. 408 p.

Zan, R. and Z. Song, 1979 Comparative analysis of karyotypes of Ctenopharyngodon idellus and Megalobrama amblycephala. Acta genet. sin. 6(2):205-210.

Anon., 1999, Fish collection database of the Natural History Museum, London (formerly British Museum of Natural History (BMNH)). Natural History Museum, London (formerly British Museum of Natural History (BMNH)).
Anon., 2001Fish collection database of the National Museum of Natural History (Smithsonian Institution). Smithsonian Institution - Division of Fishes.

Walker, K.F. and H.Z. Yang, 1999, Fish and fisheries in western China. p. 237-278. In T. Petr (ed.) Fish and fisheries at higher altitudes: Asia. FAO Fish. Tech. Pap. No. 385. FAO, Rome. 304 p.

Anon., 2002, Fish collection database of the American Museum of Natural History American Museum of Natural History, Central Park West at 79th Street, New York, NY 10024-5192, USA

Table 1. Diseases common to grass carp.

Bacterial and Fungal Diseases	Parasites (Protozoan and Metazoan)
Water mold Disease (l.), Fungal diseases Columnaris Disease (l.), Bacterial diseases Water mold Disease (e.), Fungal diseases Columnaris Disease (e.), Bacterial diseases Columnaris Disease (m.), Bacterial diseases SVC, Viral diseases	Fish louse Infestation 1, Parasitic infestations (protozoa, worms, etc.) Myxidium Infection, Parasitic infestations (protozoa, worms, etc.) Trichodina Infection 1, Parasitic infestations (protozoa, worms, etc.) Trichodina Infection 2, Parasitic infestations (protozoa, worms, etc.) Trichodina Infection 3, Parasitic infestations (protozoa, worms, etc.) Tripartiella Infestation, Parasitic infestations (protozoa, worms, etc.) Bothriocephalus Infestation 2, Parasitic infestations (protozoa, worms, etc.) Trichodina Infection 5, Parasitic infestations (protozoa, worms, etc.) Trichodina Infection 5, Parasitic infestations (protozoa, worms, etc.) Myxobolus Infection 1, Parasitic infestations (protozoa, worms, etc.) Fish louse Infestation 1, Parasitic infestations (protozoa, worms, etc.) Dactylogyrus Gill Flukes Disease, Parasitic infestations (protozoa, worms, etc.) Trichodinosis, Parasitic infestations (protozoa, worms, etc.) Sporozoa-infection (Myxobolus sp.), Parasitic infestations (protozoa, worms, etc.) Anchorworm Disease (Lernaea sp.), Parasitic infestations (protozoa, worms, etc.) Capillaria Infestation, Parasitic infestations (protozoa, worms, etc.) Philometra Disease, Parasitic infestations (protozoa, worms, etc.) Spiroxys Infestation, Parasitic infestations (protozoa, worms, etc.)

[13]***Polyodon spathula*** (Walbaum, 1792)

Common Name: paddlefish

Size: FishBase lists the maximum recorded length of paddlefish as 221 cm (male/unsexed; FishBase Ref. 5723) and a maximum weight as 90.7 kg (Ref. 3221).

Taxonomy and identification. The typical summary of taxonomic and identification information for paddlefish is listed in various sources, but Robison and Buchanan (1988); Page and Burr (1991); Etnier and Starnes (1993); Jenkins and Burkhead (1994); Mettee et al. (1996) provide good background on the species. Paddlefish are an extremely primitive form of fish having a skeleton of cartilage rather than bones, and in many respects, paddlefish are one of the most unique fish species in North America.

Paddlefish have few scales and their skin is smooth. Their color varies gray-blue to blackish-blue above to a pale gray to white belly and lower sides, and their highly distinctive snout is elongated and flattened into a paddle. Opercula are pointed and extend far back on the sides of the body, and their mouth is extremely large and toothless except in very young fish.

Life history and biology. Paddlefish are strictly freshwater and characteristically inhabit slow-flowing waters of large rivers. The original habitat of the paddlefish consisted of large free-flowing rivers with high concentrations of zooplankton (floating, microscopic aquatic animals). Paddlefish primarily feed on plankton and aquatic insects by swimming through the water with their mouth open and their long paddle-like snout weaving back and forth. The paddle appears to contain sensory organs that enables the paddlefish to detect concentrations of food (the paddle is not used to “dig” for food as is commonly thought). Backwater areas and tributary streams with dense concentrations of plankton are important feeding and nursery areas.

The species is demersal, generally preferring deep water, usually greater than 1.25 to 1.5 meters in

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[13]Original material prepared by Pam Fuller at USGS/BRD/FISC (October 11, 2000) and by U.S. Fish and Wildlife Service (North Dakota's federally listed endangered, threatened, and candidate species - 1995. U.S. Fish and Wildlife Service, Bismarck, ND. Jamestown, ND posted at Northern Prairie Wildlife Research Center Home Page. http://www.npwrc.usgs.gov/resource/distr/others/nddanger/nddanger.htm (Version 16JUL97). Edited and updated June to November, 2004.

depth. As a planktivore, paddlefish use their extended upper jaw, or "paddle," characteristic of the species to forage about the depths of their habitat and channel plankton into the mouth. Paddlefish present a highly disjoint native distribution with species in the family occurring in large rivers at mid-latitudes (50ºN - 30ºN) in China (*Psephurus gladius*) and in the Mississippi-Missouri River systems (*Polydon spathula*). The climate characteristic of their native distribution is temperate with average annual temperatures falling between 10 to 18ºC. The species presents relatively low resilience, with minimum population doubling times ranging from 4.5 to 14 years. Paddlefish are long-lived with a maximum age reported at 55 years.

Currently, populations of paddlefish occur in reservoirs where the fish have access to spawning areas that consist of deep, rocky rapids with swift currents. Paddlefish mature between 7 and 14 years of age, but do not spawn each year. Spawning occurs in the spring over gravel bars in swift currents at water temperatures of approximately 60º to 76º F. Successful paddlefish spawning has been positively correlated with water temperature and the length of spring flooding. Their eggs are a greenish-black color and during their first year of life paddlefish grow rapidly at over 1 inch per week. At maturity, adults reach nearly 7 feet in length and weigh 200 pounds.

Paddlefish populations have declined due to the destruction of spawning grounds, blockage of migratory movement by dams, channelization of rivers, loss of backwater habitat, stream water depletions, and pollution. Overharvest or mortalities due to commercial fishing with nets and traps may also be a leading cause of population declines. The paddlefish was formerly a candidate species for review by the U.S. Fish and Wildlife Service for official listing as an endangered or threatened, but as of February 28, 1996, the paddlefish is no longer listed as a candidate species. Regionally, however, paddlefish remains species of management concern. The species is listed as vulnerable by the IUCN (see IUCN Red List designation, C2b).

Native Range. In North America, the Mississippi River and major tributaries, including the Missouri River into Montana and the Ohio River to its headwaters, are the home of *Polydon* *spathula*. Representative point data are summarized in FishBase and reflected in Figure 17 for their distribution in North America. As such, paddlefish occur throughout the Mississippi River basin from southwestern New York to central Montana and south to Louisiana. Gulf Slope

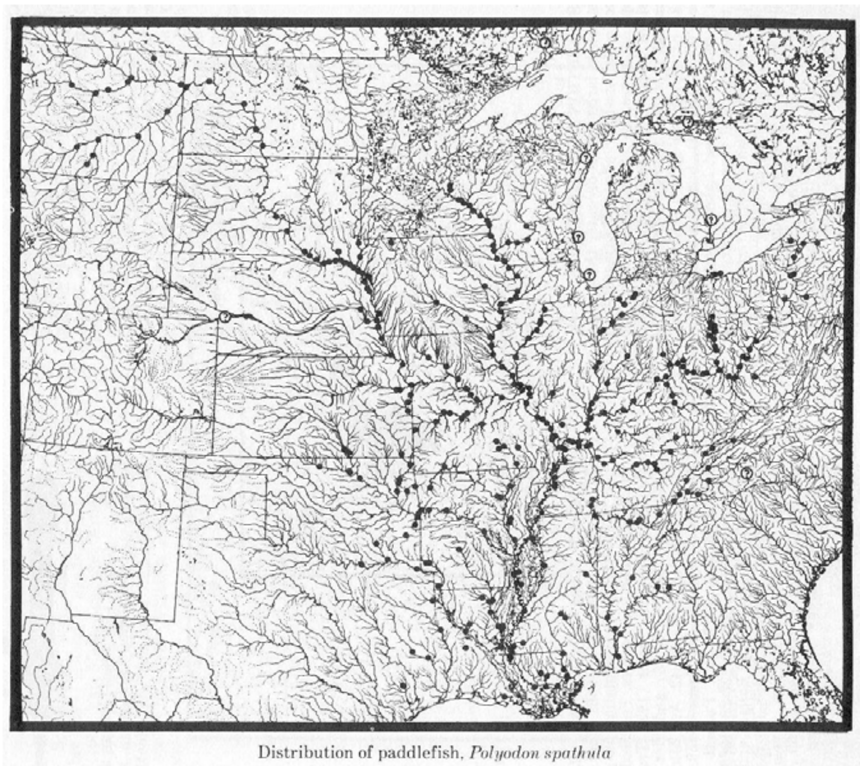
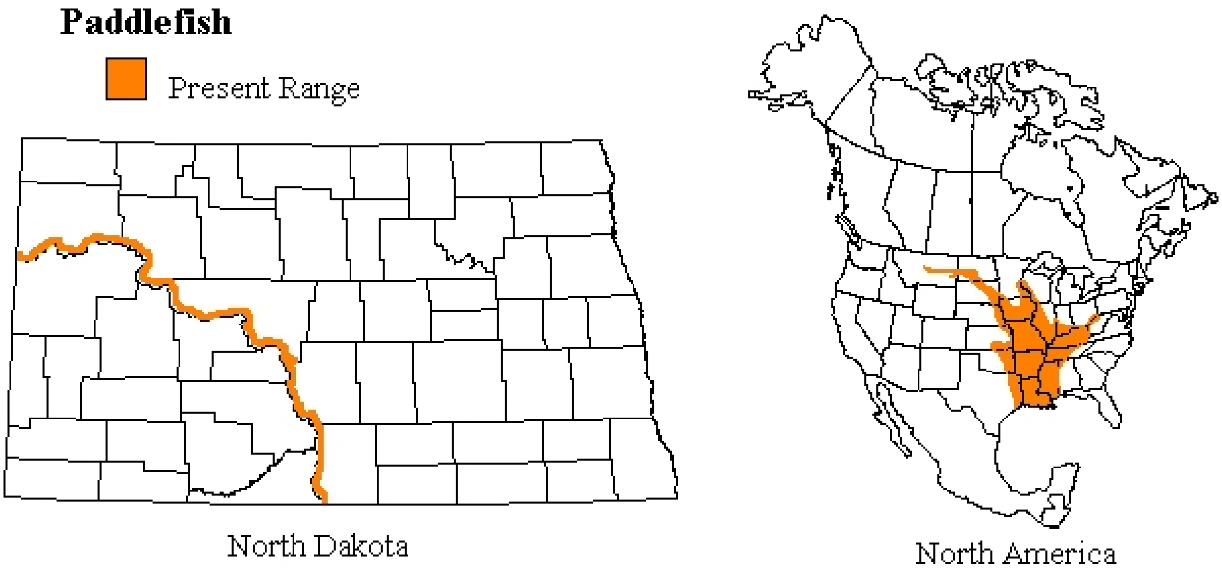


Figure 17. Point data for historic distribution of paddlefish.

drainages from Mobile Bay, Alabama, to Galveston Bay, Texas form the southern extent of the species, and formerly paddlefish occurred in the Lake Erie drainage of Ohio. Although records are incomplete, the species may have also occurred in the drainages of Lakes Huron and Michigan, but paddlefish have been extirpated from Great Lakes basin. Similarly, the species no longer occurs in drainages of Galveston Bay and Sabine River in Texas, and Calcasieu drainage in Louisiana (Page and Burr 1991).

At present, paddlefish numbers have been greatly reduced in their historic range, especially in the Mississippi and its upper tributaries. Formerly abundant in the Missouri, Mississippi and Gulf Coast drainages, paddlefish can presently be found in 22 states, including North Dakota (Figure 18). In North Dakota, they are present in the Missouri and Yellowstone Rivers. Juvenile paddlefish have been collected in North Dakota in the upper portion of Lake Sakakawea, which probably represents spawning events that most likely occur upstream in Montana with young paddlefish subsequently carried downstream into North Dakota waters.



Present distribution of the paddlefish.

Figure 18. Potential present distribution of paddlefish throughout the Mississippi River-Missouri River basins (right), and within the Missouri River basin in North Dakota (left).

Nonindigenous Occurrences. Introductions of *Polyodon spathula* have occurred worldwide during the late 20th Century (e.g., Europe, Russia, and China), although the establishment of sustainable populations is poorly characterized. *Polyodon spathula* has a long list of presence data (n = 126, see "Occurrence Records from FishBase"), but range extensions throughout the Mississippi-Missouri Rivers system (Figure 19) require field validation with respect to suspect areas maintaining sustainable populations. For example, paddlefish were introduced into the Flint River below Newton, Georgia, and fish have dispersed downstream to Lake Seminole and the Apalachicola River, Florida (R. Ober and F. Paruka, personal communication).

Means of introduction and current status. Approximately 1,200 fish, 10 to 15" in length escaped an aquaculture facility along the Flint River in Georgia, during Tropical Storm Alberto in early July 1994 (Ober and Paruka, personal communication). Four or five fish have been collected from the stretch from Newton to the Apalachicola River with one individual collected in the

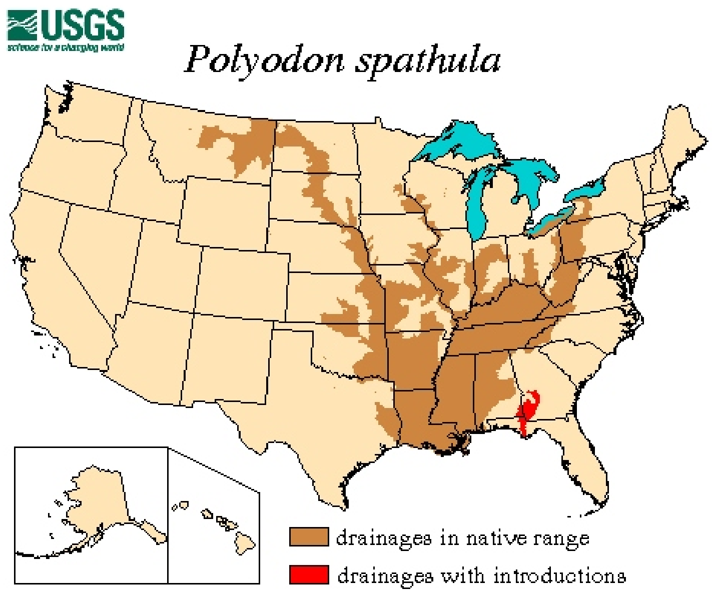


Figure 19. Drainages historically listing occurrences of paddlefish and range expansions previously noted.

spring of 1997 measuring 30" TL (Paruka, personal communication). Two paddlefish were also collected below Jim Woodruff Dam on the Apalachicola River in 1997 and weighed 10 to 11 pounds (Ober, personal communication).

Impact of Introduction. The impacts of these releases are currently unknown, however, FishBase notes that at least one country lists the species as a potential pest with adverse ecological impacts potentially expressed after introduction. International trade restrictions are in place for paddlefish (**CITES** II, since 11.6.92).

References

Adams, L.A., 1942 Age determination and rate of growth of Polyodon spathula, by means of growth rings of the otolith and dentary bone. Am. Midland Nat. 28(3):617-630.

Anon., 1996 Fish collection database of the University of British Columbia Fish Museum Fish Museum University of British Columbia, Vancouver, Canada.

Anon., 2002, Fish collection database of the American Museum of Natural History American

Museum of Natural History, Central Park West at 79th Street, New York, NY 10024-5192, USA

Anon., 2001. Fish collection database of the National Museum of Natural History (Smithsonian Institution). Smithsonian Institution - Division of Fishes.

Anon., 2000. Fish collection database of the Gulf Coast Research Laboratory (GCRL) The Gulf Coast Research Laboratory (GCRL), Ocean Springs, Mississippi, USA.

Anon., 2003 Fish Collection of the Royal Ontario Museum Royal Ontario Museum

Baillie, J. and B. Groombridge (eds.), 1996 1996 IUCN red list of threatened animals. IUCN, Gland, Switzerland. 378 p.

Birstein, V.J., 1993 Sturgeons and paddlefishes: threatened fishes in need of conservation. Conserv. Biol. 7:773-787.

Carlander, K.D., 1969 Handbook of freshwater fishery biology, volume 1. The Iowa State University Press, Ames. Iowa.

Dillard, J, K. Graham, and T. Russell. 1986. The Paddlefish: Status, Management and Propagation. Published by the North Central Division of the American Fisheries Society.

Dingerkus, G. and W.M. Howell, 1976 Karyotypic analysis and evidence of tetraploidy in the North American paddlefish, Polyodon spathula. Science 194(4267):842-844.

Echmeyer, W.N., 1997 PISCES. Eschmeyer's PISCES database as published on the Internet in June 1997, URL: gopher://gopher.calacademy.org:640/7.

Epifanio, J.M., J.B. Koppelman, M.A. Nedbal and D.P. Philipp, 1996 Geographic variation of paddlefish allozymes and mitochondrial DNA. Trans. Am. Fish. Soc. 125(4):546-561.
546-561

Etnier, D. A., and W. C. Starnes. 1993. The fishes of Tennessee. University of Tennessee Press, Knoxville, TN.

Hilton-Taylor, C., 2000, IUCN red list of threatened species. IUCN, Gland, Switzerland and Cambridge, UK. xviii + 61 p. (with 1 CD-ROM)

Houser, A., 1965 Growth of paddlefish in Fort Gibson Reservoir, Oklahoma. Trans. Am. Fish. Soc. 94(1):91-93.

Hugg, D.O., 1996, MAPFISH georeferenced mapping database. Freshwater and estuarine fishes of North America. Life Science Software. Dennis O. and Steven Hugg, 1278 Turkey Point Road, Edgewater, Maryland, USA.

International Union for Conservation of Nature and Natural Resources (IUCN), 1994, IUCN red list of threatened animals. International Union for Conservation of Nature and Natural Resources, Gland, Switzerland and Cambridge, U.K.

Jenkins, R. E., and N. M. Burkhead. 1994. Freshwater fishes of Virginia. American Fisheries Society, Bethesda, MD.

Kroll, K.J., J.P. van Eenennaam, S.I. Doroshov, J. Linares, E.J. Hamilton and T.R. Russell, 1994 Growth and survival of paddlefish fry raised in the laboratory on natural and artificial diets. Prog. Fish-Cult. 56:169-174.

Mero, S.W. , D.W. Willis and G.J. Power, 1994 Walleye and sauger predation on paddlefish in Lake Sakakawea, North Dakota. N. Am. J. Fish. Manage. 14(1):226-227.

Mettee, M. F., P. E. O'Neil, and J. M. Pierson. 1996. Fishes of Alabama and the Mobile Basin. Oxmoor House, Inc. Birmingham, AL. 820 pp.

McClanes, A.J. (ed.), 1974 Field guide to freshwater fishes of North America. Holt, Rinehart and Winston, New York. 212 p.

Ober, R. - Georgia Department of Natural Resources, Albany, GA.

Page, L.M. and B.M. Burr, 1991 A field guide to freshwater fishes of North America north of Mexico. Houghton Mifflin Company, Boston. 432 p.

Page, L. M., and B. M. Burr. 1991. A field guide to freshwater fishes of North America north of Mexico. The Peterson Field Guide Series, volume 42. Houghton Mifflin Company, Boston, MA.

Paruka, F. - USFWS, Panama City, FL.

Pflieger W.L. 1975. The Fishes of Missouri. Published by the Missouri Department of Conservation.

Reed, B.C., W.E. Kelso and D.A. Rutherford, 1992 Growth, fecundity, and mortality of paddlefish in Louisiana. Trans. Am. Fish. Soc. 121(3):378-384.

Robins, C.R., R.M. Bailey, C.E. Bond, J.R. Brooker, E.A. Lachner, R.N. Lea and W.B. Scott. 1991. World fishes important to North Americans. Exclusive of species from the continental waters of the United States and Canada.. Am. Fish. Soc. Spec. Publ. (21):243 p.

Robins, C.R., R.M. Bailey, C.E. Bond, J.R. Brooker, E.A. Lachner, R.N. Lea and W.B. Scott, 1980 A list of common and scientific names of fishes from the United States and Canada. Am. Fish. Soc. Spec. Publ. (12)1-174.

Robison, H. W., and T. M. Buchanan. 1988. Fishes of Arkansas. University of Arkansas Press, Fayetteville, AR.

Rosen, R.A., D.C. Hales and D.G. Unkenholz, 1982 Biology and exploitation of paddle fish in the Missouri River below Gavins Point Dam. Trans. Am. Fish. Soc. 111(2):216-222.

Rosen, R.A. and D.C. Hales, 1981 Feeding of paddlefish, Polyodon spathula. Copeia 1981(2):441-455.

Sola, L., S. Cataudella and E. Capanna, 1981 New developments in vertebrate cytotaxonomy III. Karyology of bony fishes: A review. Genetica 54:285-328.

[14]***Scaphirhynchus albus* (Forbes & Richardson, 1905)**

Common name: Pallid sturgeon

Maximum size. FishBase recorded a maximum length as 168 cm TL (male/unsexed), and pallid sturgeon can weigh up to 80 pounds (for comparison, shovelnose sturgeon which overlaps pallid sturgeon's range reaches a maximum weight of 5 pounds with an average weight of 2 pounds).

Taxonomy and identification. Pallid sturgeon were not identifed as a separate species until 1905, yet the species is an ancient lineage of cartilagenous fish, As are all sturgeons, the species is armored, having lengthwise rows of "bony" plates extending dorsoventrally. Their barbels are distinctive relative to other North American sturgeon, with the length of the inner barbels (4 whisker-like appendages in front of the mouth) only about 1/2 as long as the outer barbels. The fish are grayish-white in color; hence, their common name.

Life history and biology. Pallid sturgeon are demersal, freshwater fishes ranging in mid-latitudes between 44ºN - 30ºN. Within these northern latitudes the species inhabits relatively deep channels within large rivers, and prefers turbid waters, usually occupying locations with strong currents overlaying firm sand or gravel substrates. Pallid sturgeon feed on aquatic insects, mollusks and small fishes. Pallid sturgeon do not appear to reach sexually mature until they attain 3 to 4 years of age, and both male and female sturgeon may go 3 to 10 years between spawnings. In North Dakota, pallid sturgeon spawning occurs in May or June over gravel or other hard surfaces. The eggs take 5 to 8 days to hatch. The species presents relatively low resilience with a minimum population doubling time 4.5 - 14 years. Pallid sturgeon are long lived, with individuals reaching perhaps 50 years of age. Pallid sturgeons are known to hybridize with the smaller shovelnose sturgeon.

Of the 3,550 river miles the species potentially could occupy in North America, pallid sturgeon habitats have been significantly altered. For example, on the Missouri River, impoundments have created unsuitable lake-like habitat, and channelization has yielded deep, clear channels,

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[14]Original material collected from FishBase and U.S. Fish and Wildlife Service (North Dakota's federally listed endangered, threatened, and candidate species - 1995. U.S. Fish and Wildlife Service, Bismarck, ND posted at USGS/BRD/Northern Prairie Wildlife Research Center Home Page, http://www.npwrc.usgs.gov/resource/distr/others/nddanger/nddanger.htm (Version 16JUL97). Edited and updated June through November, 2004.

alterations believed detrimental to pallid sturgeon. Commercial fishing may have also played a role in the pallid sturgeon's decline. The species is listed as endangered and near extinction throughout all or a significant portion of their range (55 Federal Register 36641; September 6, 1990)[15], and international trade in the species has been restricted since 1998 under CITES II. Pallid sturgeon is also listed as endangered on the IUCN Red List (D).

Native distribution. The range of the pallid sturgeon in North Dakota overlaps with the range of the shovelnose sturgeon. Pallid sturgeon are found in the Mississippi and Missouri Rivers and their larger tributaries (Figure 20 and Figure 21 illustrate dot map of occurrences and continuous

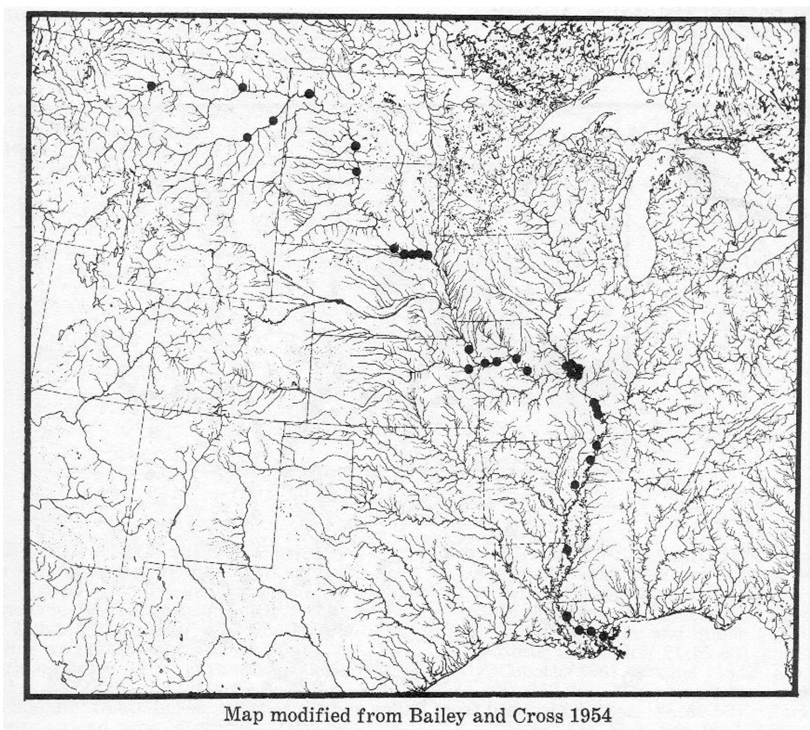
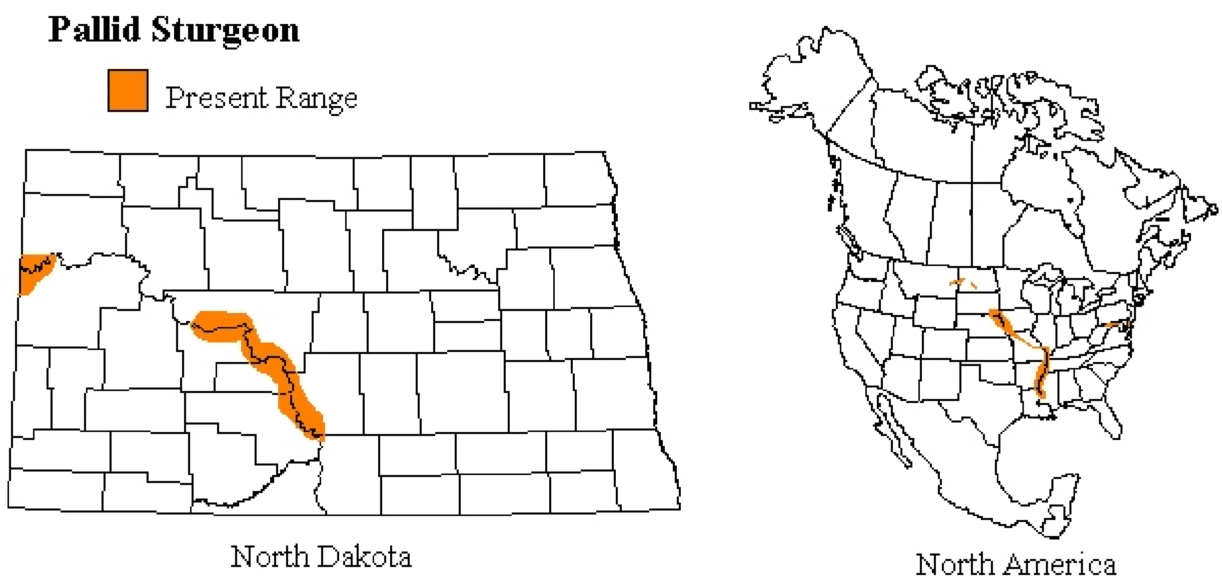


Figure 20. Dot map of locations for pallid sturgeon on the Missouri and Mississippi Rivers.

distribution map, respectively). In North America, suitable habitat is highly fragmented and restricted to main channels of Missouri River and lower Mississippi River from Montana to Louisina, and within their historic distribution, only portions of the range are presently suitable

[15]The *Pallid Sturgeon Recovery Plan* is in preparation by the North Dakota State Office of the U.S. Fish and Wildlife Service. A *Pallid Sturgeon Recovery Update* is available from the same office (1500 Capitol Ave., Bismarck, North Dakota 58501).

pallid sturgeon habitat, e.g., in North Dakota, the Missouri and Yellowstone Rivers remain relatively unaltered, but no reproduction has been documented in North Dakota in over a decade (Figure 21).



Present distribution of the pallid sturgeon.

Figure 21. Projected distribution of pallid sturgeon throughout its historic range and within North Dakota.

Non-indigenous distribution. Pallid sturgeon do not occur outside its native range where the species continues to hover near extinction with dwindling numbers of individuals.

References

Anon., 2001Fish collection database of the National Museum of Natural History (Smithsonian Institution). Smithsonian Institution - Division of Fishes.

Baillie, J. and B. Groombridge (eds.), 1996, IUCN red list of threatened animals. IUCN, Gland, Switzerland. 378 p.

Bemis, W. E., E. K. Findeis and L. Grande, 1997 An overview of Acipenseriformes. Environ. Biol. Fish. 48:25-71.

Birstein, V.J., 1993 Sturgeons and paddlefishes: threatened fishes in need of conservation. Conserv. Biol. 7:773-787.

Carlson, D. M., and Pflieger, W. L., 1981, Abundance and life history of the lake, pallid, and shovelnose sturgeons in missouri. Missouri Department of Conservation.Abstract Final Report. Endangered Species Project SE-1-10, Sturgeon Studies in Missouri, Job No. 1.

De Meulenaer, T and C. Raymakers, 1996 Sturgeons of the Caspian sea and the international trade in caviar. TRAFFIC International.

Eschmeyer, W.N., 1997 PISCES. Eschmeyer's PISCES database as published on the Internet in June 1997, URL: gopher://gopher.calacademy.org:640/7.

Fritzsch, B. and P. Moller, 1995A history of electroreception. p. 39-55. In P. Moller, Electric Fishes: history and behavior. Fish and Fisheries series 17. Chapman & Hall, London.

Hilton-Taylor, C., 2000 IUCN red list of threatened species. IUCN, Gland, Switzerland and Cambridge, UK. xviii + 61 p. (with 1 CD-ROM)

Hugg, D.O., 1996MAPFISH georeferenced mapping database. Freshwater and estuarine fishes of North America. Life Science Software. Dennis O. and Steven Hugg, 1278 Turkey Point Road, Edgewater, Maryland, USA.

International Union for Conservation of Nature and Natural Resources (IUCN), 19941994 IUCN red list of threatened animals. International Union for Conservation of Nature and Natural Resources, Gland, Switzerland and Cambridge, U.K.

Keenlyne, K.D., E.M. Grossman and L.G. Jenkins, 1992 Fecundity of the pallid sturgeon. Trans. Am. Fish. Soc. 121(1):1391-140.

Keenlyne, K.D. and L.G. Jenkins, 1993 Age at sexual maturity of the pallid sturgeon. Trans. Am. Fish. Soc. 122(3):393-396.

Keenlyne, K.D. and S.J. Maxwell, 1993 Length conversions and length-weight relations for pallid sturgeon. N. Am. J. Fish. Manage. 13:395-397.

Mayden, R.L. and B.R. Kuhajda, 1996 Systematics, taxonomy, and conservation status of the endangered Alabama sturgeon, *Scaphirhynchus suttkusi* Williams and Clemmer (Actinopterygii, Acipenseridae). Copeia 1996(2):241-273.

Page, L.M. and B.M. Burr, 1991 A field guide to freshwater fishes of North America north of Mexico. Houghton Mifflin Company, Boston. 432 p.

Robins, C.R., R.M. Bailey, C.E. Bond, J.R. Brooker, E.A. Lachner, R.N. Lea and W.B. Scott. 1991. World fishes important to North Americans. Exclusive of species from the continental waters of the United States and Canada.. Am. Fish. Soc. Spec. Publ. (21):243 p.

Robins, C.R., R.M. Bailey, C.E. Bond, J.R. Brooker, E.A. Lachner, R.N. Lea and W.B. Scott, 1980 A list of common and scientific names of fishes from the United States and Canada. Am. Fish. Soc. Spec. Publ. (12)1-174.

Robins, C.R., R.M. Bailey, C.E. Bond, J.R. Brooker, E.A. Lachner, R.N. Lea and W.B. Scott, 1991 Common and scientific names of fishes from the United States and Canada. Am. Fish. Soc. Spec. Pub. (20):183 p.

Simons, A.M., R.M. Wood, L.C. Heath, B.R. Kuhajda and R.L. Mayden, 2001 Phylogenetics of *Scaphirhynchus* based on mitochondrial DNA sequences. Trans. Am. Fish. Soc. 130:359-366.

[16]***Stizostedion lucioperca* (Linneaus 1758)**

Common Name: zander, pike-perch

Size: The usual maximum length varies between 90 cm to 1.1 m with weights between 5 to 7 kg. The largest zander recorded was 1.3m and weighed 12 kg.

Taxonomy and identification. Various authors have summarized the taxonomy and identification of the species, including Berg (1949), Wheeler (1969, 1978), Maitland (1977), Howells (1992b). Zander, formerly known as *Lucioperca lucioperca* (e.g., Berg 1949), attains maximum lengths of 1.0 to 1.3 m and weights of 5 to 7 kg (Robins et al. 1991b). There are no known voucher specimens for fish occurring in the US.

Zander has small, finely toothed scales distributed over a long, torpedo-shaped body. The head is large, and powerful jaws bear several large, recurved teeth and many smaller teeth. A small, flat spine lies at the rear of the gill cover. Dorsal fin completely separated with the front part suppored by long, sharp spines, the rear part supported by branched fin rays. Tail fin is slightly forked, and anal fin is positioned well back on the tail opposite the rear dorsal. Pelvic fins are held forwards on the body close to the pectoral fins

Fish color varies from dorsal to ventral surfaces, being light olive-brown to drab brown on the back, then lightening down the sides to a cream or white belly. Color varies with water quality, with fish being darker in peaty waters and paler in turbid water. Young and small zander often have seven to 10 dusky grey bars running down their backs as far as the lateral line, but these markings fade with age. Juvenile forms have dark grey-brown fins, but the pectoral and pelvic fins are paler, sometimes with heavy brown-black spotting.

Life history and biology. In their native range, zander or perch-pike are predators of fish, including small zander, carp, crucian carp, dace, roach, rudd, bitterling, minnow, bream, silver bream, bleak gudgeon, stone loach, perch and ruffe. The wide mouth of a Zander is adopted for catching small fish, and its eyes are adapted for detecting prey in the murkiest of conditions, e.g.,

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[16] Original material prepared by Pam Fuller and Leo Nico (April 17, 2000) from the Center for Aquatic Resource Studies, which is part of the Biological Resources Division of the Geological Survey within the U. S. Department of the Interior. Edited, revised, and updated, December, 2004.

seeing in low light or turbid conditions. Small zander feed in loose shoals, but as they grow to become solitary feeders. Although feeding will occur throughout the day, foraging intensity increases at dusk and dawn, when shoals of prey are easier to approach in the changing light. Zander also feed heavily by day in turbid water. Feeding intensity declines with low water temperatures, but zander appears to feed throughout the year and only suspends feeding activities during spawning season.

In their native ranges in Europe, zander spawn in April to June in water 1 to 3 m deep. Adults congregate in suitable sites as soon as the water temperature rises above about 12ºC, with weedy sites (e.g., open reed beds) preferred breeding habitats. Fish do not pair off; instead, several males will accompany one female as she spawns, depositing their milt in mass. Each female produces an equivalent of about 150,000 eggs per kilogram body weight. Eggs are sticky, pale yellow eggs and about 1.5 mm in diameter. Eggs are deposited among weeds, boulders or in hollows in the gravel bottom, and it has been reported that spawning areas are guarded until the eggs hatch. Hatching occurs after 7 to 10 days, and 5 to 6 mm larvae feed on the remains of their yolk sacs while their fins, mouths and teeth develop. As fry, the young fish disperse and feed actively on small crustaceans and insect larvae. By the end of their first year, zander range between 15 to 20 cm in length, and at the end of the second year, fish measure 20 to 30 cm. Females grow more quickly than males after the first year. Males mature at two to three years, females at three to four years, when they will have attained an average length of 45 cm and weight of about 1.1 kg. Maximum life span is about 20 years.

Native Range. Continental Europe to western Siberia (Figure 22; Berg 1949; Robins et al. 1991b).

Nonindigenous distribution. Figure 23 identifies areas in the US where zander has been release. In the late 1970's zander was illegally stocked in ponds near Cooperstown, New York, by an individual from Germany (J. Nickum, personal communication; Courtenay et al. 1988). Hatchery-reared zander fingerlings

Figure 22. Native distribution of zander included eastern Europe (light blue), with introduction to western Europe (dark blue). Figure from Greenhalgh 1999).

were stocked into Spiritwood Lake (Stutzman County), North Dakota in 1989; the fish were imported as fry from Finland (Lohman 1989; Anderson 1992). Only one survivor was recovered during a subsequent netting survey (Anderson 1992), and a sustainable population was thought not to have been established. However, the capture of a fish in August 1999, and another 2+ year old fish in 2000 shows that at least some survived and reproduced (Dokken 2004). Concern exists that zander and walleye could hybridize.

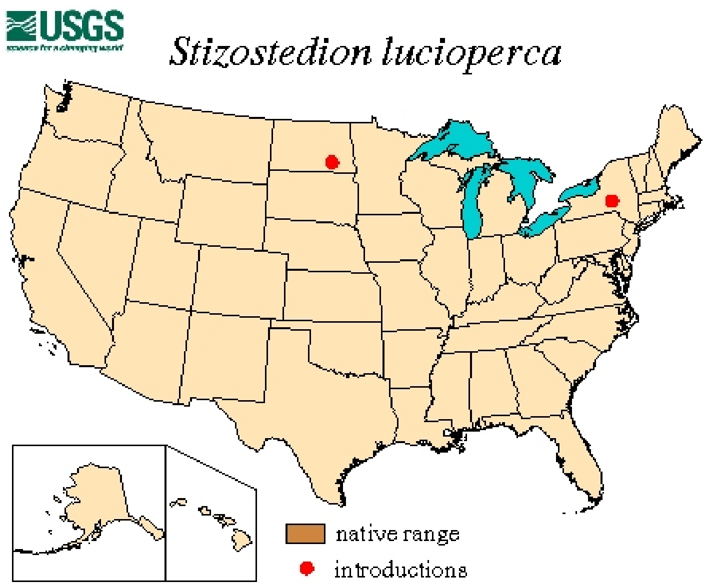

Figure 23. Distribution of known releases of zander in the US.

The history of the species introduction into North Dakota is not well documented in the scientific literature, but North Dakota Game and Fish Department had been interested in zander as a sport fish for many years. Spirit Lake had been selected as the site of an experimental release because the water body was completely enclosed (Anderson 1992). In 1987 an earlier attempt at introducing zander was aborted when fry hatched from eggs imported from Holland were destroyed for fear that they carried pike fry rhobdo virus (Anonymous 1987a; Lohman 1989). Those wanting to introduce zander thought that it would be a boon to the fisheries of North America (e.g., Anderson 1992), but others had strong reservations (e.g., Wright 1992). Some fisheries personnel in states surrounding North Dakota and nearby Canadian provinces expressed doubts concerning the species' introduction, particularly because its effect on native species was unknown and because of its potential to spread (e.g., Wingate 1992). Zander has been widely introduced into western Europe and the species was illegally introduced into portions of England.

According to Hickley (1986), the success of introduced populations seemed to be limited by the availability of the species' preferred habitat, characterized as "eutrophic, turbid, well oxygenated and of low mean depth, and, if a river, slow-flowing rather than turbulent."

Impact of Introduction: Zander stocks ore highly prized, but there is concern about the effects on native fish populations where it is introduced. Adverse effects are largely unknown, although there is concern among European fish resource managers that introduced zander may cause a collapse in resident prey fish stocks (Hickley 1986 and references therein).

References and Supplemental Bibliography (derived from FishBase)

Anderson, R. O. 1992. A case for zander: fish for the future? Pages 22-32 in The In-Fisherman Walleye Guide 1992. Brainerd, MN.

Anon., 2003 Fish Collection of the Royal Ontario Museum Royal Ontario Museum

Anon., 2002Fish collection database of the American Museum of Natural History American Museum of Natural History, Central Park West at 79th Street, New York, NY 10024-5192, USA

Anon., 1999, sh collection database of the Natural History Museum, London (formerly British Museum of Natural History (BMNH)). Natural History Museum, London (formerly British Museum of Natural History (BMNH)).

Anon., 2000, The ichthyological collection of the Zoological Museum Hamburg (ZMH) Division of Ichthyology and Herpetology, Zoological Museum Hamburg (ZMH)

Anon., 2001Fish collection database of the National Museum of Natural History (Smithsonian Institution). Smithsonian Institution - Division of Fishes.

Anonymous. 1987a. Zander program setback. Dakota Country, July 1987.

Berg, L. S. 1948-1949. Freshwater fishes of the U.S.S.R. and adjacent countries, 4th edition. Three volumes. Translated from Russian, 1962-1965, for the Smithsonian Institution and the National Science Foundation, by Israel Program for Scientific Translations, Jerusalem, Israel.

Volume 1:504 pp.; volume 2:496 pp.; volume 3:510 pp.

Danzmann, R.G., 1979 The karyology of eight species of fish belonging to the family Percidae. Can. J. Zool. 57:2055-2060

Dokken, B. 2004. Angler's catch likely a rare zander. GrandForksHerald.com. July 13, 2004.

Eschmeyer, W.N., Editor, 2003 Catalog of fishes. Updated database version of March 2003. Catalog databases as made available to FishBase in March 2003.

Eschmeyer, W.N., 1990 Catalog of the genera of recent fishes. California Academy of Sciences, San Francisco, USA. 697 p.

Garibaldi, L., 1996 List of animal species used in aquaculture. FAO Fish. Circ. 914. 38 p.8

Greenhalgh, M., 1999, Freshwater fish, Michell Beazley, An imprint of Octopus Publishing Group, Ltd., London, UK, 192pp.

Hickley, P. 1986. Invasion by zander and the management of fish stocks. Philosophical Transactions Royal Society of London B 314:571-582.

Holcik, J., 1991Fish introductions in Europe with particular reference to its central and eastern part. Can. J. Fish. Aquat. Sci. 48 (Suppl. 1):13-23.

Howells, R. G. 1992b. Guide to identification of harmful and potentially harmful fishes, shellfishes and aquatic plants prohibited in Texas. Texas Parks and Wildlife Department Special Publication, Austin, TX. 182 pp. (+ appendices).

Kottelat, M., 1997 European freshwater fishes. Biologia 52, Suppl. 5:1-271.173

Lehtonen, H., 1987 Selection of minimum size limit for pike-perch (Stizostedion lucioperca) in the coastal waters of Finland. Proc. V Congr. Europ. Ichthyol. , Stockholm 1985 p. 351-355.

Linfield, R.S.J. and R.B. Rickards, 1979 The zander in perspective. Fish Manage. 10(1):1-16.

Lohman, J. 1989. Biologists introduce zander into North America. The Forum, Fargo, ND, Saturday, July 22, 1989.

Maitland, P.S. and R.N. Campbell, 1992 Freshwater fishes of the British Isles. HarperCollins Publishers, London.368 p.288-292

Maitland, P. S. 1977. The Hamlyn guide to freshwater fishes of Britain and Europe. Hamlyn Publishing Group Limited, New York, NY.

Mayr, B., M. Kalat, P. R b, and M. Lambrou, 1987Band karyotypes and specific types of heterochromatin in several species of European percid fishes (Percidae, Pisces). Genetica 75:199-205

Myers, R.A., J. Bridson and N.J. Barrowman, 1995 Summary of worldwide spawner and recruitment data. Can. Tech. Rep. Fish. Aquat. Sci. 2024. 274 p.

Nygren, A., P. Edlund, U. Hirsch and L. Ahsgren, 1968 Cytological studies in perch (Perca fluviatilis L.), pike (Esox lucius L.), pike-perch (Lucioperca lucioperca L.), and ruft (Acerina cernua L.) Hereditas. 59(2-3):518-524.

Pinder, A.C., 2001, Keys to larval and juvenile stages of coarse fishes from fresh waters in the British Isles. Freshwater Biological Association. The Ferry House, Far Sawrey, Ambleside, Cumbria, UK. Scientific Publication No. 60. 136 p.10, 12, 24

Robins, C. R., R. M. Bailey, C. E. Bond, J. R. Brooker, E. A. Lachner, R. N. Lea, and W. B. Scott. 1991b. World fishes important to North Americans exclusive of species from the continental waters of the United States and Canada. American Fisheries Society Special Publication 21. American Fisheries Society, Bethesda, MD. 243 pp.

Sokolov, L.I. and L.S. Berdicheskii, 1989 Acipenseridae. The freshwater fishes of Europe. Vol. 1, Part II. p. 150-44361

Stolyarov, I.A., 1985 Dietary features of catfish, Silurus glanis, and pike-perch Stizostedion lucioperca, in Kyzlyarsk Bay, northern Caspian Sea. J. Ichthyol. 25(2):140-145.141

Vehanen, T., P. Hyv„rinen and A. Huusko, 1998 Food consumption and prey orientation of piscivorous brown trout (Salmo trutta) and pikeperch (Stizostedion lucioperca) in a large regulated lake. J. Appl. Ichthyol. 14(1-2):15-22.16-17

Welcomme, R.L., 1988 International introductions of inland aquatic species. FAO Fish. Tech. Pap. No. 294. 318 p. 270-271

Wheeler, A., 1992 A list of the common and scientific names of fishes of the British Isles. J. Fish Biol. 41(1):1-37.

Wheeler, A. 1969. The fishes of the British Isles and north-west Europe. Michigan State University Press, East Lansing, MI. 613 pp.

Wheeler, A. 1978. Key to the fishes of northern Europe. Frederick Warne Ltd., London, England.

Wingate, P. J. 1992. Zander-evaluate carefully before introducing. Zander, evaluate carefully before introducing. Page 32 in The In-Fisherman Walleye Guide 1992. Brainerd, MN.

Winkler, H.M., K. Skora, R. Repecka, M. Ploks, A. Neelov, L. Urho, A. Gushin and H. Jespersen, 2000 Checklist and status of fish species in the Baltic Sea. ICES CM 2000/Mini:11, 15 p.

Wright, D. 1992. Zander hot line. Newsletter of the Introduced Fish Section, American Fisheries Society 11(3):2.

Representative biota of concern: Aquatic, wetland, and terrestrial vascular plants

[17]***Hydrilla verticillata* (L.f.) Royle (Hydrilla)**

Common name: hydrilla

Taxonomy and identification. Hydrilla grows submersed. The species is generally perennial but sometimes annual, and have horizontal stems in the substrate forming tubers under certain conditions. Stems are ascending and usually are sparsely branched until the plants near the water surface and then become profusely branched. Under certain conditions, turions (actually bulbil-like structures) form in the leaf axils. Stems can be up to 8.5 m long and grow to the surface of the water where the branchlets extend horizontally. Leaves are 1-nerved, sessile, whorled, 3 to 12 at a node but mostly 5 or more, mostly shorter than 1.5 cm long, linear to lanceolate or rarely widely ovate, broadest at the base, the sides nearly paralleling to near the acute tip that terminates in a single spine cell. Leaf margins are serrate, the teeth visible to the naked eye. Fresh leaves are notably rough to the touch. The midrib on the upper surface is often tinged with red and on the lower surface, usually, has 1-celled sharp teeth or spines.

Flowers are unisexual, arising from the leaf axil; plants are monoecious or dioecious. The flowers are small, less than 6 mm in diameter, translucent to white; female flowers are usually produced in the fall and are on long thread-like stalks 2 to 4 cm long from leaf axils of the upper branches that carry the flowers to the water surface. Male flowers are solitary, small, on short stalks in the leaf axil and break off as buds, opening explosively on the water surface.

Hydrilla can usually be differentiated from Canadian elodea (*Elodea canadensis* Michx.) and egeria (*Egeria densa* Planch.) by the following characters:

- Leaves mostly in whorls of 4 at sterile nodes, leaves 1.4 to 2.5 cm long *Egeria densa*
- Leaves of stems at growing tips at water's surface usually in whorls of 3 or 5 or more; leaves not or mostly not exceeding 1.5 cm long, sometimes to 2 cm

[17]Original material prepared by C.C. Jacono and M.M. Richerson at the Center for Aquatic Resource Studies, USGS/BRD/FISC. Last accessed December, 2004; edited and revised December, 2004.

- Leaves mostly in whorls of 5 or more; margins of the leaves with teeth perceptible to the naked eye; midribs on lower leaf surface (when fresh) with a few conical protuberances tipped by sharp 1-celled teeth; fresh leaves notably rough to the touch *Hydrilla verticillata*
- Leaves mostly in whorls of 3; margins of the leaves not having teeth perceptible to the naked eye; midribs of lower leaf surface not pronounced, not bearing teeth; fresh leaves not rough to the touch *Elodea canadensis*

Habitat and growth characteristics. Hydrilla grows in canals, springs, streams, ponds, lakes and reservoirs. Most populations of hydrilla in the United States are dioecious, although populations of monoecious hydrilla occur in North Carolina and northward into the mid-Atlantic states (Langeland 1996). Hydrilla can reproduce by four methods: fragmentation, tubers, turions, and seed. Tubers in the hydrosoil can remain viable for several years (Langeland 1996) and allow the plant to survive cold temperatures and periods of drought (Tarver *et al.*1986). Although the importance of seed production in the spread of hydrilla has not been researched extensively, it is probably of minor importance compared to vegetative reproduction (Langeland 1996). Hydrilla has a high growth rate and lower light requirement for photosynthesis than most other submersed plants (Langeland 1996) which allows it to grow at greater depths and outcompete most other species. It also forms a dense canopy at the surface of the water and "shades out" other submersed plants (Tarver *et al*. 1986).

Native distribution and introduction to North America. *Hydrilla verticillata*(L.f.) Royle is an introduction from the Old World (Cook & Luond 1982) that was first discovered in the United States in 1960. The species is now abundantly naturalized in many parts of the United States (Langeland 1996). Plants have attractive foliage and are planted in aquaria which are often emptied into freshwater habitats. Hydrilla is easily confused with *Egeria densa* Planch., Brazilian elodea or Egeria, and *Elodea canadensis* Michx., Canadian elodea, Waterweed.

Non-indigenous distribution. The current distribution of hydrilla in the US is illustrated in Figure 24.

Control and management problems.[18] This species is probably the worst submersed aquatic

[18]Developed in part from guidance from The Western Aquatic Plant Management Society available from http://www.wapms.org/plants/hydrilla.html last accessed December, 2004.

weed in the United States. Plants form large, dense populations which displace native species, restrict flow, and impair small boat navigation and other recreational uses (Tarver *et al*. 1986, Langeland 1996). In addition to being spread by natural fragmentation, plants are sometimes spread from lake to lake by fragments attached to boat motors and trailers. Hydrilla also is thought to be intentionally introduced into "new" water bodies in an effort to enhance sport fishing for black bass.

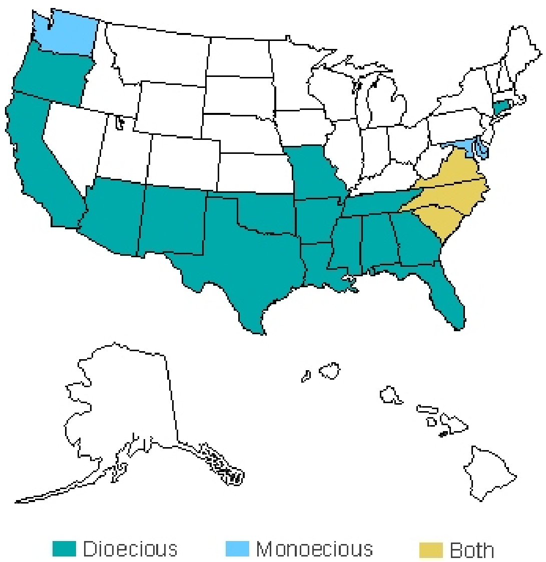

Figure 24a. Distribution of hydrilla as indicated by state records.

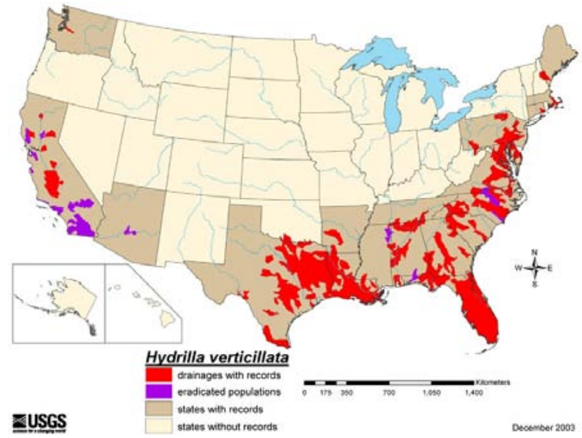

Figure 24b. Distribution of hydrilla as indicated by hydrological unit codes.

Management of hydrilla in waterways or ponds and lakes is generally intensive, owing to hydrilla's great reproductive potential. Tubers are particularly troublesome, since they serve as a source of regrowth in areas where the hydrilla shoots have been controlled by chemical or mechanical methods. Hydrilla can rapidly colonize an area devoid of aboveground vegetation.

Response to Herbicides. Three EPA-registered herbicides are effective against hydrilla growth that are permitted for use: fluridone (Sonar®), endothall (Aquathal®), and copper compounds. Fluridone is a systemic herbicide that has proven effective against hydrilla; however, drawbacks to using fluridone include its high cost, slow-action, and non-selectivity toward other macrophyte species. Endothall, a fast-acting contact herbicide, is used when immediate control of vegetation is needed. Copper compounds are often used in conjunction with endothall applications, although copper by itself exhibits herbicidal action against hydrilla. Copper is also used for its algicidal properties when heavy periphytic growth on the hydrilla may interfere herbicide uptake. These herbicides do not affect hydrilla seeds, tubers, and turions and repeated applications are needed to control hydrilla regrowth.

Response to Cultural Methods. Localized control (in swimming areas and around docks) can be achieved by covering the sediment with a opaque fabric which blocks light from the plants. Managers of reservoirs and some lake systems may have the ability to lower the water level as a method of managing aquatic plants. This technique is sometimes successful in areas where the hydrosoil can thoroughly desiccate.

Response to Mechanical Methods. Because this plant spreads readily through fragmentation, mechanical controls such as cutting and harvesting should be used only when the infestation has reached a local maximum (e.g., dispersal is resource limited). Using mechanical controls while the plant is still invading, will tend to enhance its rate of spread.

In some states, e.g., Florida, specially designed aquatic plant harvesters are used to cut and collect hydrilla from waterways. Hydrilla harvesting is mainly performed to open boat lanes through hydrilla beds for navigation. Because hydrilla produces more biomass per square meter than most aquatic plants, the cost of harvesting hydrilla is generally higher than for harvesting other nuisance species such as Eurasian watermilfoil. For example, harvesting costs on the Potomac River were about $1,200 per acre (costs for harvesting milfoil in Washington average $600 to 800 per acre).

Biological Control. Worldwide surveys for natural hydrilla enemies were begun in 1981 in a cooperative study between the University of Florida, the United States Department of Agriculture, and the U.S. Army Corps of Engineers. A number of insects were identified, quarantined and tested, and eventually released in Florida and other states. Results from these insect releases are still being evaluated.

Although they have access to many biocontrol agents, grass carp have been deemed the most effective biological control for hydrilla by Florida lake managers. Grass carp have definite food preferences and hydrilla is a preferred plant species. Grass carp have proven to be an effective tool for hydrilla eradication, e.g., hydrilla infestation reached a maximum of 79 percent on Lake Baldwin, Florida, and was then eliminated by two successive grass carp stockings. Similarly, in California triploid grass carp have been introduced to the Imperial Irrigation District as an eradication tool for hydrilla.

References

Cook, C. D. K. and R. Luond. 1982. A revision of the genus *Hydrilla* (Hydrocharitaceae). Aquatic Botany 13:485-504.

Langeland, K. A. 1996. *Hydrilla verticillata* (L.f.) Royle (Hydrocharitaceae), the perfect aquatic weed. Castanea 61(3):293-304.

Tarver, D. P., J. A. Rogers, M. J. Mahler, and R. L. Lazor. 1986. Aquatic and Wetland Plants of Florida. Third Edition. Florida Department of Natural Resources, Tallahassee, Florida.

[19]***Myriophyllum spicatum*** L.

Common Name: Eurasian watermilfoil

Taxonomy and identification. A submersed, rooted, perennial herb. Consisting of long underwater stems that branch and produce many whorled, finely divided leaves upon nearing the surface. Distinguished from the native *M. sibiricum* primarily by the overall shape of the leaf and then by the number of leaflets. Leaves are divided in to threadlike leaflets, usually in pairs of more than 14 (Nichols 1975). Leaflets are uniformly tapered so that the leaf shape is more like an equilateral triangle with a curved base. Leaflets stand at acute angles (less than 45 degrees) to the rachis and are parallel to each other (Ceska 1985). Meanwhile, *M. sibiricum* has basal leaflets that are as long as the leaf. They curve over and extend almost to the top of the leaf, forming a more feathery shape. Aiken (1981) provides a detailed key for fertile specimens.

Life history and biology. Lakes, ponds, shallow reservoirs and low energy areas of rivers and streams. Brackish water of protected tidal creeks and bays. Particularly troublesome in waterbodies that have experienced disturbances such as nutrient loading, intense plant management, or abundant motorboat use (Nichols 1994).

Native distribution. Europe, Asia, and northern Africa

Nonindigenous Occurrences: Figure 25 illustrates the current distribution of Eurasian watermilfoil in the US. In the northeastern US, the species is expanding through New York, particularly into the Upper Hudson River-Albany region and into lakes in the foothills and mountains of the Adirondacks (Madsen 1994). In New England, Eurasian watermilfoil occurs in Vermont at 53 lakes, most concentrated in the western drainages, where *Myriophyllum spicatum* covers thousands of aquatic acres, including large bays in Lake Champlain and Lake Bomoseen (Crosson 2000). Eradication has been accomplished in some areas of New England; for example, the species has been successfully eradicated from the interior of New Hampshire by draining a site (Mountain Pond) in Brookfield, New Hampshire (R. Esterbrook, New Hampshire Dept. of Environmental Services, pers. comm. 1996). However, Eurasian watermilfoil has since found in

[19] Original material prepared by Colette C. Jacono and M.M. Richerson (April 15, 2003) from the Center for Aquatic Resource Studies which is part of the Biological Resources Division of the Geological Survey within the U.S. Department of the Interior. Updated January, 2004.

the Connecticut River, bordering Vermont and New Hampshire, and under New Hampshire jurisdiction (Engel 1998). The species is locally abundant and spreading rapidly in Massachusetts and Connecticut (Crow and Hellquist 1983), and occurs in lakes and ponds of Rhode Island

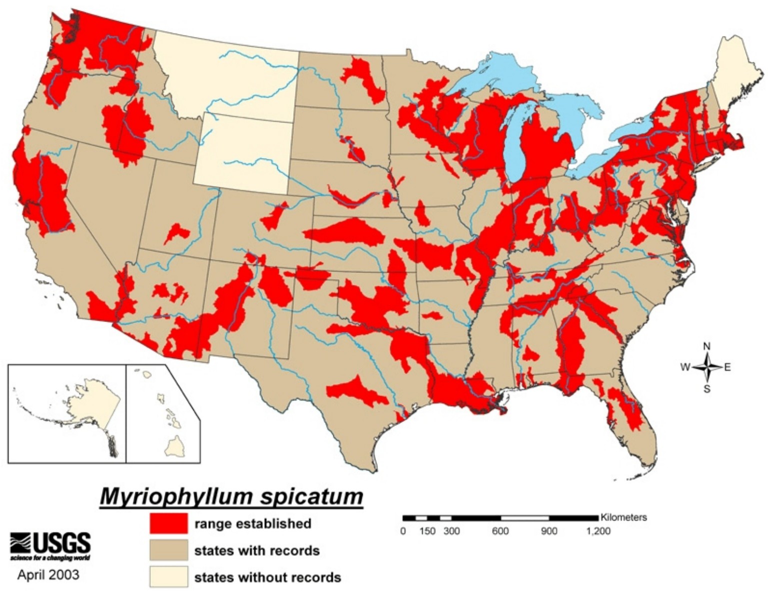

Figure 25. Map indicates recorded presence in at least one site within the drainage (USGS Hydrologic Unit 8) , but does not necessarily imply occurrence throughout that drainage.

(Sheath and Nerone 1988). The species is spreading rapidly through lakes and rivers in Pennsylvania, while the native *M. sibiricum* has been listed as endangered (Pennsylvania Flora Project 1998). Watermilfoil has become established in the tidal regions of the Delaware River, where salt intrusion and industrial pollution are eliminating native submersed plants (Schuyler et al. 1993). The species occurs in New Jersey's Upper Delaware drainage basin where specimens were collected as early as 1952 from Lake Musconetcong (Schuyler 1989), and now occurs in all major drainages in New Jersey. In New Jersey, Eurasian watermilfoil is very problematic in the state's northern lakes (Trudeau 1982), and the species is recently been observed in Delaware from a pond along the Chesapeake & Delaware Canal (C. Martin, Delaware Dept. of Natural Resources and Environmental Control, pers. comm. August 1997). In Maryland, the species is common in fresh to oligohaline waters of the Upper Chesapeake Bay and its tributaries (Orth et al. 1996).

In the southeast, Eurasian watermilfoil has occurred in the Potomac River esturary for over three decades, varying in abundance depending on location and seasonal influences in grow and vigor of plants in Virginia (Carter and Rybicki 1994). In North Carolina, the species has been declining in some waters; for example, Eurasian watermilfoil once dominated the shallow waters of Currituck Sound, but climatic factors and sediment suspension precipitated lead to the species' decline in 1990 (Carter and Rybicki 1994). Depending on site conditions, the species has covered at least 4000 acres in the Currituck and Albermarle Sounds, and has become established inland at Lake Gaston and the adjoining Roanoke Rapids Lake, North Carolina (NCDWR 1996). In neighboring South Carolina, Eurasian watermilfoil has been known since 1972 at a few public lakes (Lake Murray and Stevens Creek reservoirs; S. deKozlowski, South Carolina Dept. of Natural Resources, pers. comm. 1997). At Lake Seminole, Georgia, Eurasian watermilfoil was replaced by hydrilla following extensive 2,4-D treatment (Bates and Smith 1994), but the species remains widespread in private impoundments throughout Georgia (G. Lewis, Univ. of Georgia, pers. comm. 1999). Eurasian watermilfoil often dominates the vegetation community in the lower portions of the Apalachicola, Homosassa, Chassohowitzka and Crystal Rivers of Florida, where they meet the Gulf of Mexico (BAPM 1982-1994). Similar patterns of invasion occur throughout the Gulf Coast. In Alabama, Eurasian watermilfoil is the most abundant submersed species in bays and creeks of the Mobile River Delta (Zolczynski and Shearer 1997), and the species has occurred in freshwater reservoirs throughout the rest of the state for years (Bayne 1979). In Mississippi, Eurasian watermilfoil has occurred along the Tennessee-Tombigbee waterway since 1987 (Kight 1988).

West of the Appalachians, Eurasian watermilfoil occurs in West Virginia (Harmon et al 1996), and in Kentucky, populations of the species have declined in some areas (e.g., Kentucky Lake), while being maintained as large populations at other impoundments throughout the state (B. Kenman, Kentucky Fish and Wildlife, pers. comm. 1996). Populations have fluctuated in size throughout the the Tennessee River system (Smith and Barko 1996) where the species was initially introduced to the river at Watts Bar Reservoir, Tennessee in the 1950's (Couch and Nelson 1985). Eurasian watermilfoil has been spreading throughout the Cumberland River system in the late 1980's, in some instances as the result of deliberate planting (Simpson 1990).

In the Great Lakes Region, Eurasian watermilfoil is decreasing in some locations, e.g., Put-in-Bay Harbor, Lake Erie, Ohio, while native plants have returned, as nonindigenous mussels clear the once turbid waters of the harbor (Stuckey and Moore 1995). In the glacial lakes of Indiana, Eurasian watermilfoil has been repeatedly found to occur, with many of the sightings recorded for lakes located in the northern St. Joseph drainage (INDNR 1997). The species is also known in reservoirs across central Indiana (IDNR 1997), including Monroe Reservoir, where the species thrives in silt laden zones (Landers and Frey 1980). In Illinois, the species has been declining in McCullom Lake in conjunction with the appearance of herbivorous *Euhrychiopsis* weevils (Weinberg 1995). Eurasian watermilfoil is well established in Wisconsin, where infestations have occurred in nutrient rich, recreational lakes since the 1960's. Southern Wisconsin has recorded large populations of Eurasian watermilfoil for over 40 years, and Wisconsin records the most occurrences of the species for any state (over 300 waterbodies infested; Engel 1999). In Lake Michigan the plant occurs sporadically in some bays, but it is spreading northward through lakes of Michigan's lower peninsula (Nichols 1994; Trudeau 1982). Eurasian watermilfoil has been spreading rapidly in Minnesota since the species' arrival in 1987 (e.g., Eurasian watermilfoil occurs in 75 lakes and 4 streams that radiate from the Twin Cites area (Bratager et al. 1996).

Across the Great Plains Eurasian watermilfoil is increasingly becoming a problem. The species is a relatively recent arrival to North Dakota, where it was initially observed in September 1996 as local populations in the Sheyenne River at Valley City (B. Alexander, Valley City State Univ., pers. comm. 1997). The species recurs sporadically but its occurrence is influenced by environmental conditions, e.g., the species was not found following flooding and drawdown of 1997 (Engel 1998). In South Dakota , Eurasian watermilfoil was recently observed at Lake Sharpe, a 61,010 acre impoundment of the Missouri River, and the species has been observed from Fort Thompson to Pierre, where a few small beds were found in August 1999 (D. Ode, South Dakota Game, Fish and Parks, pers. comm. 2000). The species is rare and of little concern in most of Nebraska, populations vary across the state (e.g., noted as declining at Wildwood Lake, Lancaster County while increasing in Hord Lake, Merrick County; R. Kaul, Univ. of Nebraska, pers. comm. 1997; T. LeGrange, Nebraska Game and Parks, pers. comm. 1996). In Kansas, reports of local populations are limited, but the species is long standing along the Kerr-McClellan waterway and in ponds and lakes of southern and central Oklahoma (Nelson and Couch 1985). Since 1993, the plant has occurred as various locations sites across Iowa, where nutrient loading, sedimentation and the maintenance of artificially high water levels have contributed to the absence of native vegetation. The species is currently established at Wilson Grove Lake and at Snyder Bend Lake, a shallow oxbow of the Missouri River (G. Phillips, Iowa Lakes Community College, and J. Wahl, Iowa Dept. of Natural Resources, pers. comm. 1997).

In the Ozark-Prairie of south central US, Eurasian watermilfoil has been collected since 1962 at various locations within 9 river drainages in Missouri (Padgett 2001). The plants are most problematic in the southcentral and southeastern portions of the state, especially at major recreational water bodies including Lake of the Ozarks (Whitley et al. 1990) and the upper Gasconade River (Padgett 2001). In Arkansas, the species has been tentatively identified from vegetative specimens collected at Lake Ouachita (herbarium specimen UARK 1997), and the species probably occurs downstream in Lake Hamilton (M. Armstrong, Arkansas Dept. of Game and Fish, pers. comm. 1996). The species is found locally in lakes and bayous of western and southeastern Louisiana, and more commonly occurs in fresh to brackish marshes and bays of the Mississippi Delta and the southern Coastal zone (Montz 1980; Chabreck and Condrey 1979). Populations have been established at reservoirs in eastern and central Texas where the species is most troublesome at Lake Austin, Pat Mayse Reservoir and Buescher State Park (Helton and Hartmann 1996).

Throughout the west and northwest US, occurrence of Eurasian watermilfoil is sporadic. Early sightings of *Myriophyllum spicatum* from Montanta and Wyoming are not well documented (Engel 1998), but are likely the result of misidentification. However, the species has been observed in 1998 at the Pend Oreille River, Hayden Lake, Spirit Lake and Eagle Island State Park, in western Idaho (C. Holly and V. Mason, Idaho Dept. of Agric., pers. comm. 1998), which represents the first records for the state. These observations coincide with warm summers in the Pacific Northwest. In Oregon, Eurasian watermilfoil occurs in Devils Lake, where efforts to managed the plant have relied on releases of triploid grass carp (*Ctenopharyngodon idella*; Bonar et al. 1993). The species occurs in lakes across Washington and along the Columbia, Little Spokane, and Pend Orielle Rivers (Parsons 1996) where the invasive plant has replaced native vegetation, particularly in shallow lakes east of Puget Sound, Washington (Walton 1996).

In the intermountain west and southwest US, Eurasian watermilfoil was initially reported in 1998 from Colorado from the Rio Grande River near Alamosa (F. Nibling, U.S. Bureau of Reclamation, pers. comm. 1999) where its presence has caused concern for regional irrigation systems. In New Mexico, the species has been reported at ponds and lakes in four northern counties (Martin and Hutchins 1981), and more recent records have been noted for Abiquiu and Cochiti Lakes, impoundments on the Rio Chama and the Rio Grande Rivers (Charles Ashton, U.S. Army Corps of Engineers, pers. comm. 2000). In Arizona as in other areas of the southwest, Eurasian watermilfoil may be more a curiosity than a problem, given the warm, arid climate of the region. In Arizona, the speices occurs in a few ponds in the Colorado River Indian Tribe Reservation and in a small reservoir in the Verde Valley (E. Hall, Arizona Dept. of Agriculture, pers. comm. 1997). First observed in 1993, Eurasian watermilfoil occurs at Fish Lake and Otter

Creek Reservoir in Utah (UDWR 1993), and in Nevada, plants were first observed at marinas along the northern shore of Lake Tahoe in 1995 (Anderson and Ryan 1996). And, in California, the species occurs as an nncommon plant in ditches and at lake margins in regions surrounding San Francisco Bay and San Joaquin Valley (Hickman 1993).

Means of Introduction. In the US, Eurasian watermilfoil was first documented in 1942 from a pond in Washington D.C., where the species was probably intentionally introduced (Couch and Nelson 1985). From that point and other introduction, the species has spread into lakes and streams across the country. Once introduced, water currents potentially disseminate vegetative propagules throughout a drainage, and stem fragments are important for the colonization of new habitats. Within a stand, local populations generally expand through growth of stolons (Aiken et al. 1979; Madsen et al. 1988). Anthropogenic mechanisms such as motorboat traffic contribute to natural seasonal fragmentation and the distribution of fragments throughout lakes, and transport of watercraft plays the largest role in introducing fragments to new waterbodies. For example, road checks in Minnesota have found aquatic vegetation on 23% of all trailered watercraft inspected (Bratager 1996) where transport of any aquatic vegetation is now illegal.

Impact of introduction. Eurasian watermilfoil is one of the most widely distributed of all nonindigenous aquatic plants in the US, being confirmed in 45 states. In Canada, the species occurs British Columbia, Ontario and Quebec. The species is widespread primarily because it competes aggressively to displace and reduce the diversity of native aquatic plants, once introduced to an area. From a single to a few vegetative propagules, the plant becomes established, then its shoots elongate and vegetative growth continues. The plant effectively outcompetes native aquatic vegetation, since its growth in spring occurs much earlier than other aquatic plants. The species is tolerant of low water temperatures, and it quickly grows to the surface forming dense canopies that overtop and shade the surrounding vegetation (Madsen et al. 1991). Canopy formation and light reduction are significant factors in the decline of native plant abundance and diversity observed when Eurasian water-milfoil invades healthy plant communities (Smith and Barko 1990; Madsen 1994). Both eelgrass (*Vallisneria americana*) and southern naiad (*Najas guadalupensis*) are known to have been displaced by this nonindigenous species in the Mobile Delta of Alabama (Bates and Smith 1994), and its establishment in Lake George, New York, reduced native plants from 5.5 to 2.2 species per square meter, in just two years (Madsen et al 1991). Eurasian water-milfoil has less value as a food source for waterfowl than the native plants it replaces (Aiken et al. 1979). Fish may initially experience a favorable edge effect in early stages of Eurasian watermilfoil invasions, but the characteristic over abundant growth offsets any short-term benefits that edge effect may provide fish in healthy waters. At high densities, the plant's foliage supports a lower abundance and diversity of invertebrates, organisms that serve as fish food (Keast 1984). Dense cover allows high survival rates of young fish, but larger predator fish lose foraging space and and are less efficient at obtaining their prey (Lillie and Budd 1992; Engel 1995). Madsen et al. (1995) found growth and vigor of a warm-water fishery reduced by dense Eurasian water-milfoil cover. The growth and senescence of thick vegetation degrades water quality and depletes dissolved oxygen levels (Honnell 1992; Engel 1995). Typical dense beds restrict swimming, fishing and boating, clog water intakes and result in decaying mats that foul lakeside beaches.

Control and management. The occurrence of sixteen species including *Potamogeton illinoensis* and *Potamogeton pectinatus* may be indicators of conditions suitable for Eurasian water-milfoil invasion. Searching areas colonized by these species may provide early detection, the best method for preventing new invasion (Nichols and Buchan 1997).

Chemical and physical control.[20] Most problems caused by milfoil can be managed with conventional methods such as treatment with herbicides or mechanical removal of plants. For example, in Minnesota, management of Eurasian watermilfoil is completed using methods that cause as little damage to native aquatic plants as possible. Native plants provide many benefits to lake ecosystems, such as stabilizing lake sediments, and increasing habitat for fish and wildlife. Also, widespread destruction of native plants can lead to an overall increase in the amount of Eurasian watermilfoil in a water body because milfoil is very effective at invading disturbed habitat.

Using herbicides to eradicate milfoil are "...rarely, if ever, likely to succeed" (Smith and Barko 1990), and in Minnesota, the use of herbicides to prevent the spread of milfoil within a lake has, at best, slowed the plant's local dispersion, but rarely yields eradication. Studies by Minnesota Department of Natural Resources (DNR) suggests that fluridone application to whole lakes or bays at an intermediate rate of 10 parts per billion (ppb) causes unavoidable damage to native vegetation and has the potential to affect other aspects of lake ecosystems (Welling et al 1997).

The results of subsequent investigations of fluridone in Michigan suggested that application of the herbicide at the low rate of 5 to 6 ppb may provide more selective control than has previously

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[20]Source from http://www.dnr.state.mn.us/ecological_services/invasives/ewmprog.html.

been observed in Minnesota. Chemical control of Eurasian watermilfoil is variously successful, and should be considered along with other agents and management tools as indicated by case-specific factors.

For example, in the Washington state,[21][22] various eradication and control strategies have been developed to address a wide range of invasive plant management issues. These management tools include eradication and control strategies using physical and chemical controls, e.g., hand pulling and bottom barrier installation; diver dredging; rotovation and harvesting; water level drawdown; 2,4-D treatment; whole lake fluridone treatment, endothall treatment; and release of triploid grass carp.

Due to expense and the time intensive nature of manual methods, sites suitable for hand pulling and bottom screening are limited to lakes or ponds only lightly infested with Eurasian watermilfoil. This method is suitable for very early infestations of milfoil and for follow-up removal after a whole lake fluridone treatment, a 2,4-D treatment, or diver dredging. To be cost-effective, generally the total amount of milfoil in the waterbody should be three-acres or less in area, if all the milfoil plants were grouped together in one location. If the infestation has advanced beyond this point, it is more effective to consider other eradication techniques such as aquatic herbicides. This method may also be applicable in waterbodies where no herbicide use can be tolerated such as in a lake used as a municipal drinking water supply. These methods could be used in any waterbody to eradicate milfoil; however costs for large-scale projects would become prohibitive.

Response to Herbicides. Westerdahl and Getsinger (1988) reported excellent control with 2,4-D, diquat, diquat and complexed copper, endothall dipotassium salt, and endothall with complexed copper. They also report good control with fluridone, and in Washington, fluridone (Sonar®) has been successfully used to eradicate Eurasian watermilfoil in lakes. To be effective, fluridone concentrations of 10-15 ppb must be maintained in the water column for 10 to 12 weeks. Follow-up diver surveillance and hand-pulling of surviving plants is essential to the success of this technique. Some eradication attempts with fluridone have had mixed success in Washington. Factors such as surface and ground water inflows and development of land forms of Eurasian

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[21]See http://www.ecy.wa.gov/programs/wq/plants/management/milfoil_strategies.html.

[22]See http://www.wapms.org/plants/milfoil.html.

watermilfoil all affect the success rate. The herbicide triclopyr is undergoing federal aquatic registration and holds great promise for Eurasian watermilfoil control. Unlike fluridone, triclopyr requires a short contact time (18 to 48 hours) and will selectively control Eurasian watermilfoil while leaving many native aquatic plants relatively unaffected.

Response to Cultural Methods. Localized control (in swimming areas and around docks) can be achieved by covering the sediment with a opaque fabric which blocks light from the plants (bottom barriers or screens). Managers of reservoirs and some lake systems may have the ability to lower the water level as a method of managing aquatic plants. The Tennessee Valley Authority (TVA) uses both winter and summer water level drawdowns as effective way of reducing Eurasian watermilfoil biomass. Drawdown of about 2 meters is effective in reducing excessive populations. Short-term dewatering for 2-3 days during period of freezing temperatures has been effective, but multiple exposures may improve control. A 1-week drawdown of a large TVA impoundment in July 1983 desiccated about 810 hectares of Eurasian watermilfoil. A narrow, relatively weed-free band occurred after refilling and control effects extended into the following two growing seasons. In Washington, the Bureau of Reclamation lowered the water level of Banks Lake in 1994 in an effort to manage Eurasian watermilfoil populations. The success of a drawdown on Eurasian watermilfoil is dependent on several factors such as degree of desiccation (drawdowns in rainy western Washington and Oregon are often ineffective), the composition of substrate (sand vs. clay), air temperature (the exposed sediments need to freeze down to 8-12 inches), and presence of snow.

Response to Mechanical Methods. Because Eurasian watermilfoil spreads readily through fragmentation, mechanical controls such as cutting, harvesting, and rotovation (underwater rototilling) should be used only when the extent of the infestation is such that space for further expansion is limited and, e.g., local within-lake dispersion is unlikely. Using mechanical controls while the plant is still invading, will tend to enhance its rate of spread. The British Columbia Ministry of Environment developed a barge mounted rototilling machine called a rotovator to remove Eurasian watermilfoil roots. The machine's underwater tiller blades churn up to 8 inches into the sediment and dislodge buoyant Eurasian watermilfoil roots. Floating roots may then be collected from the water. Control with rotovation, generally extends 2 or more growing seasons.

Harvesting requires machinery that can cut plants below the water's surface, then collected, and stored harvested plant material for disposal on land. Harvesting removes surfacing mats and creates open areas of water. However because of its rapid growth rate Eurasian watermilfoil generally needs to be harvested twice during the growing season.

Biological control. A range of biological control agents have been used in efforts to eradicate or control Eurasian watermilfoil. Insects have been evaluated by USDA and Corps of Engineers, and several insects have been considered as prospective biocontrol agents, e.g., pyralid moths and several stem-boring weevils. However, many of these insects were found to be non-specific to Eurasian watermilfoil or to offer little potential as effective biological control agents. In British Columbia, several insects were associated with Eurasian watermilfoil and a midge was investigated as a potential control agent. However, the midge proved to be extremely difficult to rear in the laboratory.

A North American weevil, *Euhrychiopsis lecontie*, may be associated with natural declines at northern lakes (Sheldon 1994, Bratager et al. 1996, Weinberg 1995). Studies have found the herbivorous weevil to cause significant damage to Eurasian water-milfoil while having little impact on native species (Sheldon and Creed 1995), suggesting the insect as a potential biological control agent. For example, *E. lecontei* has been found in Washington state feeding on both Eurasian watermilfoil and northern milfoil (*Myriophyllum sibericum*) plants, and studies have shown that this native weevil appears to be a milfoil specialist and will not feed on other macrophyte species. It can be easily raised in the laboratory and laboratory-reared weevils could be used to augment natural populations.

Although triploid grass carp will eat Eurasian watermilfoil, it is not a highly palatable or preferred species. To achieve control of Eurasian watermilfoil generally means the total removal of more palatable native aquatic species before the grass carp will consume Eurasian watermilfoil. In situations where Eurasian watermilfoil is the only aquatic plant species in the lake, this may be acceptable. However, generally grass carp are not recommended for Eurasian watermilfoil control.

Plant pathogens of Eurasian watermilfoil may be applicable to control and management of Eurasian watermilfoil, especially given observations of extensive mortality of Eurasian watermilfoil linked to a plant pathogenic fungus *Mycoleptodiscus terrestris*. In the late 1960s in Maryland, “Northeast Disease” was associated with declines in Eurasian watermilfoil, and *M. terrestris* was suspected as the causative agent. The pathogenic fungus has been shown to significantly reduce Eurasian watermilfoil biomass in laboratory studies, although control Eurasian watermilfoil in the field has not been achieved. The US Army Corps of Engineers is continuing research on plant pathogens.

References

Aiken, S.G., P.R. Newroth and I. Wile. 1979. The biology of Canadian weeds. 34. *Myriophyllum spicatum* L. Canadian Journal of Plant Science 59:201-215.

Anderson, L.W. J. and F.J. Ryan. 1996. Eurasian watermilfoil in Lake Tahoe: a threat to a national treasure. Pp. 18-19 in Abstracts Thirty-sixth Annual Meeting of the Aquatic Plant Management Society, Inc. July 14-17 1996, Burlington, VT.

(BAPM) Bureau of Aquatic Plant Management. 1982-1994. Florida Aqautic Plant Management Surveys, electronic data. Bureau of Aquatic Plant Management, Florida Department of Environmental Protection, Tallahassee, Florida.

Bates, A.L., and C.S. Smith. 1994. Submersed plant invasions and declines in the Southeastern United States. Lake and Reservoir Management 10(1):53-55.

Bayne, D.R. 1979. The most troublesome aquatic weeds of Alabama. Proceedings of the Southern Weed Science Society 32:280-283.

Bode, J, S. Borman, S. Engel, D. Helsel, F. Koshere, and S. Nichols. 1993. Eurasian Water Milfoil in Wisconsin: A Report to the Legislature. Wisconsin Department of Natural Resources, Madison, WI. 36 pp.

Bonar, S.A., G.L. Thomas, S.L. Thiesfeld, G.B. Pauley, and T.B. Stables. 1993. Effect of triploid grass carp on the aquatic macrophyte community of Devil's Lake, Oregon. North American Journal Fisheries Management. 13(4):757-765.

Bratager, M., W. Crowell, S. Enger, G. Montz, D. Perleberg, W.J. Rendall, L. Skinner, C.H. Welling and D. Wright. 1996. Harmful Exotic Species of Aquatic Plants and Wild Animals in Minnesota. Annual Report. Minnesota Department of Natural Resources, St. Paul, MN. 99 pp.

Brewer, C., and M. Parker. 1990. Adaptations of macrophytes to life in moving water: upslope limits and mechanical properties of stems. Hydrobiologia 194:133-142.

Carpenter, S.R. and D.M. Lodge. 1986. Effects of submersed macrophytes on ecosystem process. Aquatic Botany 26:341-370.

Ceska, O. and A. Ceska. 1985. *Myriophyllum*Haloragaceae species in British Columbia: problems with identification. Pp. 39-50 in: L.W.J. Anderson [ed.] Proceedings of the First International Symposium on Watermilfoil (*Myriophyllum spicatum* and related Haloragaceae species. Aquatic Plant Management Society, Vicksburg, Mississippi.

Chabreck, R.H., and R. E. Condrey. 1979. Common Vascular Plants of the Louisiana Marsh. Sea Grant Publication No. LSU-T-79-003. Louisiana State University Center for Wetland Resources, Baton Rouge, LA.

Cohen, A.N., and J.T. Carlton. 1995. Nonindigenous Aquatic Species In A United States Estuary: A Case Study Of The Biological Invasions Of The San Francisco Bay and Delta. A Report for the United States Fish and Wildlife Service, Washington D.C.

Couch, R., and E. Nelson. 1985. *Myriophyllum spicatum*in North America. Pp. 8-18 in L.W.J. Anderson (ed.). First International Symposium Watermilfoil and Related Haloragaceae Species. 23-24 July 1985, Vancouver, B.C. Aquatic Plant Management Society, Vicksburg, MS.

Crosson, H. 2000. Vermont Aquatic Plant Survey. Vermont Department of Environmental Conservation, Waterbury, VT.

Crow, G.E., and C.B. Hellquist. 1983. Aquatic Vascular Plants of New England: Part 6. Trapaceae, Haloragaceae, Hippuridaceae. New Hampshire Agricultural Experiment Station, University of New Hampshire, Durham, NH.

Engel, S. 1999. Eurasian watermilfoil database. Wisconsin Department of Natural Resources, Woodruff, Wisconsin.

Engel, S. 1995. Eurasian watermilfoil as a fishery management tool. Fisheries 20(3):20-27.
Harmon, P.J., J.T. Kartesz, C.M. Jessee, B.R. McDonald, and B.D. Sargent. 1996. Checklist of the Wetland and Vascular Plants of West Virginia. The West Virginia Natural Heritage Program, West Virginia Division of Natural Resources, Elkins, WV. 74 pp.

Helton, R. J., and L. H. Hartmann. 1996. Statewide Aquatic Vegetation Survey Summary, 1995 Report. Statewide Freshwater Fisheries Monitoring and Management Program, Texas Parks and Wildlife Department, Austin, TX.

Hickman, J.C. (ed.) 1993. The Jepson Manual: Higher Plants of California. University of California Press, Berkeley, CA.

Honnell, D., J.D. Madsen, and R.M. Smart. 1992. Effects of aquatic plants on water quality in pond ecosystems. Proceedings: 26th Annual Meeting, Aquatic Plant Control Research Program. Report A-92-2. U.S. Army Corps of Engineers Waterways Experiment Station, Vicksburg, MS.
(IDNR) Indiana Department of Natural Resources. 1997. Exotic Plant Species In Indiana Lakes. Lake and River Enhancement Program, Division of Soil Conservation, Indiana Department of Natural Resources, Indianapolis, IN.

Keast, A. 1984. The introduced aquatic macrophyte, *Myriophyllum spicatum*, as habitat for fish and their macroinvertebrate prey. Can. J. Zool. 62:1289-1303.

Kight, J. 1988. Tennessee-Tombigbee Waterway. Proceedings: 22nd Annual Meeting, Aquatic Plant Control Research Program. Report A-88-5. U.S. Army Corps of Engineers Waterways Experiment Station, Vicksburg, MS.

Landers, D.H. and D.G. Frey. 1980. The Dieback Role of *Myriophyllum spicatum* in Monroe Reservoir, Indiana. Technical Report No. 134, Office of Water Research Technology, U.S. Department of the Interior.

Lillie, R.A., and J. Budd. 1992. Habitat architecture of *Myriophyllum spicatum* L. as an index to habitat quality for fish and macroinvertebrates. Journal of Freshwater Ecology 7(2):113-125.

Madsen, J.D. 1994. Invasions and declines of submersed macrophytes in Lake George and other Adirondack lakes. Lake and Reservoir Management 10(1):19-23.

Madsen, J.D., L.W. Eichler, and C.W. Boylen. 1988. Vegetative spread of Eurasian watermilfoil in Lake George, New York. Journal of Aquatic Plant Management 26:47-50.

Madsen, J.D., J.W. Sutherland, J.A. Bloomfield, L.W. Eichler, and C.W. Boylen. 1991. The decline of native vegetation under dense Eurasian watermilfoil canopies. J. Aquatic Plant Management 29:94-99.

Madsen, J.D., R.M. Smart, G.O. Dick, and D.R. Honnell. 1995. The influence of an exotic submersed aquatic plant, *Myriophyllum spicatum*, on water quality, vegetation, and fish populations of Kirk Pond, Oregon. Proceedings: 29th Annual Meeting, Aquatic Plant Control Research Program. US Army Corps of Engineers Waterways Experiment Station.

Martin, W.C. and C. R. Hutchins. 1981. A Flora of New Mexico. Vol. 2. J. Cramer. pp. 1403-1405.

Montz, G. 1980. Distribution of selected aquatic plant species in Louisiana. The Proceedings of the Louisiana Academy of Sciences 43:119-138.

Nelson, E.N., and R.W. Couch. 1985. Aquatic Plants of Oklahoma I: Submersed, Floating-leaved and Selected Emergent Macrophytes. Oral Roberts University, Tulsa, OK.

NCDWR (North Carolina Division of Water Resources) 1996. Economic and Environmental Impacts of N.C. Aquatic Weed Infestations. Department of Environment, Health and Natural Resources, NC.

Nichols, S. A. 1975. Identification and management of Eurasian Water Milfoil in Wisconsin. Wisconsin Academy of Sciences, Arts and Letters 63:116-126.

Nichols, S. A. 1994. Evaluation of invasions and declines of submersed macrophytes for the Upper Great Lakes Region. Lake and Reservoir Management 10(1):29-33.

Nichols, S.A. and L.A. Buchan. 1997. Use of native macrophytes as indicators of suitable Eurasian watermilfoil habitat in Wisconsin lakes. Journal of Aquatic Plant Management 35:21-24.

Orth, R.J., J.F. Nowak, G.F. Anderson, D.J. Wilcox, J.R. Whiting, and L.S. Nagey. 1996. Distribution of Submerged Aquatic Vegetation in the Chesapeake Bay and Tributaries and Chincoteague Bay - 1995. U.S. Environmental Protection Agency, Annapolis, MD.

Parsons, J.P. 1996. Aquatic Plant Technical Assistance Program: 1995 Activity Report.

Washington State Department of Ecology, Environmental Investigations and Laboratory Services Program, Olympia, WA.

Padgett, D.J. 2001. Noteworthy collections and spread of exotic aquatics in Missouri. Castanea 66(3):303-306.

Pennsylvania Flora Project. 1998. http://www.upenn.edu/paflora. Botany Department, Morris Arboretum, University of Pennsylvania, Philadelphia, PA.

Rabe, F.W., and S.W. Chadde. 1994. Classification of aquatic and semiaquatic wetland natural areas in Idaho and Western Montana. Natural Areas Journal 14:175-187.

Sheath, R.G. and C.A. Nerone. 1988. Lake and Pond Littoral-Zone Macrophytes. Ch. 4 in: R.G. Sheath and M.M. Harlin [ed.] Freshwater and Marine Plants of Rhode Island. Kendall Hunt Publishing Co., Dubuque, IA.

Sheldon, S.P. 1994. Invasions and declines of submersed macrophytes in New England, with particular reference to Vermont Lakes and herbivorous invertebrates in New England. Lake and Reservoir Management 10(1):13-17.

Sheldon, S.P., and R. P. Creed. 1995. Use of a native insect as a biological control for an introduced weed. Ecological Applications 5(4):1122-1132.

Schuyler, A.E., S.B. Andersen, and V.J. Kolaga. 1993. Plant zonation changes in the tidal portion of the Delaware River. Proceedings of The Academy of Natural Sciences of Philadelphia. 144:263-266.

Schuyler, A.E. 1989. Submerged vascular plants in the Delaware River from the Delaware Water Gap to Trenton. Bartonia 55:53-58.

Smith, C.G., and J.W. Barko. 1990. Ecology of Eurasian Watermilfoil. Journal of Aquatic Plant Management 28:55-64.

Smith, C.G., and Barko, J.W. 1996. Evaluation of a *Myriophyllum spicatum*decline in reservoirs of the Tennessee and Cumberland rivers. Technical Report A-96-6, U.S. Army Engineer

Waterways Experiment Station, Vicksburg, MS.

Stuckey, R.L., and D. L. Moore. 1995. Return and increase in abundance of aquatic flowering plants in Put-In-Bay Harbor, Lake Erie, Ohio. Ohio Journal of Science 95(3):261-266.

Trudeau, P. 1982. Nuisance Aquatic Plants and Aquatic Plant Management Programs in the United States. Volume 3: Northeastern and North Central Region. MITRE Corporation, McLean, VA.

(UDWR) Utah Division of Wildlife Resources. 1993. More on Eurasian Watermilfoil. Utah Wildlife, Division of Wildlife Resources, Department of Natural Resources, UT. 3pp.
(UARK) University of Arkansas Herbarium, Fayetteville, AR.

Walton, S.P. 1996. Aquatic Plant Mapping for 36 King County Lakes. King County Surface Water Management Division, Seattle, WA.

(WDNR) Wisconsin Department of Natural Resources. 1990. Environmental Assessment Aquatic Plant Management (NR 107) Program, WI. 213 pp.

Whitley, J.R, B. Bassett, J. Dillard, and R. Haefner. 1990. Water Plants for Missouri Ponds. Missouri Department of Conservation, Jefferson City, MO. 151 pp.

Zolczynski, J. and R. Shearer. 1997. Mobile Delta Submersed Aquatic Vegetation Survey, 1994. Fisheries Section, Game and Fish Division, Alabama Department of Conservation and Natural Resources, Spanish Fort, AL.

[23][24]***Eichhornia crassipes*** (Martius) Solms

Common name: Waterhyacinth

Taxonomy and identification. Taxonomy of waterhyacinth is detailed in on Cronquist (1988), Thorne (1992), and Takhtajan (1997). The English common names of the plant are waterhyacinth, water hyacinth, and water-hyacinth. Waterhyacinth is the standardized spelling adopted by the Weed Science Society of America (WSSA, 1984) to denote that it is not an aquatic relative of true "hyacinth" (*Hyacinthus* spp.), as the two-word spelling suggests.

Waterhyacinth is a member of the pickerelweed family (Pontederiaceae). Families most closely allied with the Pontederiaceae are Commelinaceae, Haemodoraceae (including Conostylidaceae [Takhtajan, 1997]), Philydraceae, and Hanguanaceae (Hahn, 1997; APG, 1998). The subclass Commelinidae includes the Arecales, Poales, Commelinales, and Zingiberales (APG, 1998). The Pontederiaceae is a small family of herbaceous mono Cotyledons that includes six genera and 30 to 35 species (Eckenwalder and Barrett, 1986). All are palustrine or aquatic and most are confined to the Americas. All seven members of the genus *Eichhornia* originated in tropical America, except for *Eichhornia natans* (P. Beauv.), which is from tropical Africa. Fourteen species of Pontederiaceae occur in the flora of US and Canada (Table 2), six of which are adventive; none are considered threatened or endangered (USDA, NRCS, 1999).

Life history and biology. Waterhyacinth is an erect, free-floating, stoloniferous, perennial herb. The bouyant leaves vary in size and morphology. The short, bulbous leaf petioles produced in uncrowded conditions provide a stable platform for vertical growth. Plants in crowded conditions form elongate (up to 1.5 m) petioles (Center and Spencer, 1981). Leaves are arranged in whorls

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[23] Original material prepared by T. D. Center (U.S. Department of Agriculture, Agricultural Research Service, Invasive Plant Research Laboratory, Fort Lauderdale, Florida, USA, M. P. Hill - Plant Protection Research Institute, Pretoria, South Africa, H. Cordo - U.S. Department of Agriculture, Agricultural Research Service, South American Biological Control Laboratory, Hurlingham, Argentina, M. H. Julien - Commonwealth Scientific and Industrial Research Organization, Indooroopilly, Australia, and published in Van Driesche, R., *et al.*, 2002, Biological Control of Invasive Plants in the Eastern United States, USDA Forest Service Publication FHTET-2002-04, 413 p.

[24]Supplemental life history derived from Michael S. Batcher, 1907 Buskirk-West Hoosick Rd., Buskirk, New York, and John M. Randall and Barry Meyers-Rice, The Nature Conservancy, Wildland Invasive Species Program, Department of Vegetable Crops and Weed Science, 124 Robbins Hall, University of California, Davis, California.

of six to 10, and individual plants develop into Clones of attached rosettes (Center and Spencer, 1981).

Table 2. Species of Pontederiaceae in the United States.

Native Species	Introduced Species
Heteranthera dubia (Jacq.) MacM.	*Eichhornia azurea* (Sw.) Kunth
Heteranthera limosa (Sw.) Willd.	*Eichhornia crassipes* (Mart.) Solms.
Heteranthera mexicana Wats.	*Eichhornia diversifolia* (Vahl) Urban
Heteranthera multiflora (Griseb.) Horn	*Eichhornia paniculata* (Spreng.l) Solms
Heteranthera penduncularis Benth.	*Monochoria hastata* (L.) Solms
Heteranthera reniformis Ruiz Lopez & Pavon	*Monochoria vaginalis* (Burm. f.) K. Presl
Heteranthera rotundifolia (Kunth) Griseb.	
Pontederia cordata L.	

The lavender flowers display a central yellow fleck and are borne in clusters of up to 23 on a single spike (Barrett, 1980). The flowers may have short, medium, or long styles, but only the short- and long-style forms occur in the United States (Barrett, 1977). The 14-day flowering cycle concludes when the flower stalk bends, positioning the spike below the water surface where seeds are released (Kohji *et al.*, 1995). Seed capsules normally contain fewer than 50 seeds each (Barrett, 1980). Each inflorescence can produce more than 3,000 seeds and a single rosette can produce several inflorescences each year (Barrett, 1980). The small, long-lived seeds sink and remain viable in sediments for 15 to 20 years (Matthews, 1967; Gopal, 1987). Seeds germinate on moist sediments or in warm shallow water (Haigh, 1936; Hitchcock *et al.*, 1950) and flowering can occur 10 to 15 weeks thereafter (Barrett, 1980). Lack of germination sites limits seedling recruitment except during drought, on decaying mats after herbicide applications (Matthews, 1967), or at the margins of waterbodies. Populations increase mainly by vegetative means.

Weber (1950), Richards (1982), Watson (1984), and Watson and Cook (1982, 1987) describe waterhyacinth growth and population expansion as the result of differentiation of apical or axillary meristems. The single apical meristem on each stem tip can be vegetative, producing leaves with axillary buds, or reproductive, producing flowers. If an inflorescence develops, termination of the apical meristem halts leaf production. In this event, the axillary bud immediately below the inflorescence differentiates into a continuation shoot. This produces a new apical meristem that allows leaf production to proceed. If the axillary bud does not form a continuation shoot, then it produces a stolon. Elongation of the stolon internode moves the axillary bud apex away from the parent rosette. It then produces short internodes that grow vertically into a new rosette.

Waterhyacinth grows best in neutral pH, water high in macronutrients, warm temperatures (28º to 30ºC), and high light intensities. It tolerates pH 4.0 to 10.0 (Haller and Sutton, 1973), but not more than 20 to 25% sea water (Muramoto *et al.*, 1991). The plants survive frost if the rhizomes do not freeze, even though emergent portions may succumb (Webber, 1897). Prolonged cold kills the plants (Penfound and Earle, 1948), but reinfestation from seed follows during later warmer periods. Ueki (1978) matched the northern limit of waterhyacinth to the 1ºC average January isotherm in Japan, and growth is inhibited at water temperatures above 33˚C (Knipling *et al.*, 1970). Plants stranded on moist sediments can survive several months (Parija, 1934).

Native distribution. The diversity of other species of *Eichhornia*, particularly the more primitive *Eichhornia paniculata* (Spreng.) Solms. and *Eichhornia paradoxa* (Mart.) Solms., and the overlapping range of the closely related *Eichhornia azurea* (Sw.) Kunth suggest that *E. crassipes* arose in tropical South America.

Non-indigenous distribution. Waterhyacinth was introduced into the United States around 1884 and has become pan-tropical. Worldwide, the limits of distribution are bound by 40ºN and 40ºS latititude (Gowanloch and Bajkov, 1948; Bock, 1968; Holm *et al.*, 1969; Ueki, 1978; Gopal, 1987). In the US, waterhyacinth is most abundant in the Southeast (Figure 26); it also occurs in California and Hawaii, with scattered records in other states (USDA, NRCS, 1999).

Impacts and control. Waterhyacinth, *Eichhornia crassipes* is one of the world's worst weeds (Holm *et al.*, 1977), invading lakes, ponds, canals, and rivers. It was introduced into many countries during the late 19th and early 20th centuries, where it spread and degraded aquatic ecosystems. The species is still rapidly spreading throughout Africa, where new infestations are creating life-threatening situations as well as environmental and cultural upheaval (Cock *et al.*, 2000). Control with herbicides, particularly 2,4-D, is feasible, but is costly and temporary.

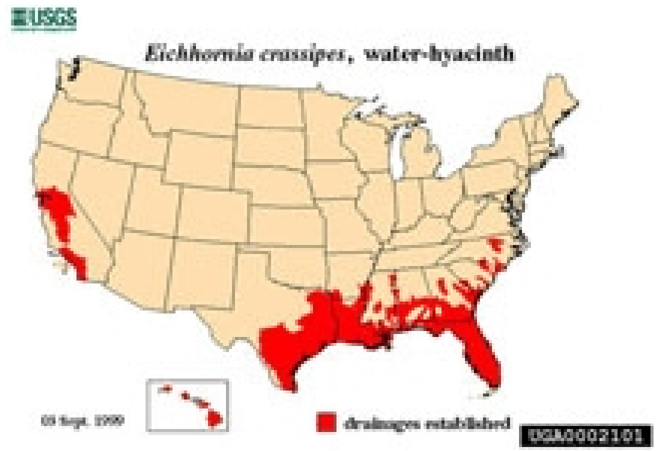

Figure 26. Drainages infested by waterhyacinth in the United States. (Map from USGS/BRD).

Adverse biological and ecological effects associated with waterhyacinth are numerous. Dense mats reduce light to submerged plants, thus depleting oxygen in aquatic communities (Ultsch, 1973). The resultant lack of phytoplankton (McVea and Boyd, 1975) alters the composition of invertebrate communities (O'Hara, 1967; Hansen *et al.*, 1971), ultimately affecting fisheries. Drifting mats scour vegetation, destroying native plants and wildlife habitat. Waterhyacinth also competes with other plants, often displacing wildlife forage and habitat (Gowanloch, 1944). Higher sediment loading occurs under waterhyacinth mats due to increased detrital production and siltation. Herbicidal treatment or mechanical harvesting of waterhyacinth often damages nearby desirable vegetation.

Economic impacts of waterhyacinth may be significant. Waterhyacinth grows rapidly (Penfound and Earle, 1948) forming expansive colonies of tall, interwoven floating plants that may blanket large waterbodies, and create impenetrable barriers and obstructing navigation (Gowanloch and Bajkov, 1948; Zeiger, 1962). Floating mats block drainage, causing flooding or preventing subsidence of floodwaters. Large rafts accumulate where water channels narrow, sometimes causing bridges to collapse. Waterhyacinth hinders irrigation by impeding water flow, by clogging irrigation pumps, and by interfering with weirs (Penfound and Earle, 1948). Multimillion-dollar flood control and water supply projects can be rendered useless by waterhyacinth infestations (Gowanloch and Bajkov, 1948).

Infestations block access to recreational areas and decrease waterfront property values, oftentimes harming the economies of communities that depend upon fishing and water sports for revenue. Shifting waterhyacinth mats sometimes prevent boats from reaching shore, trapping the occupants and exposing them to environmental hazards (Gowanloch and Bajkov, 1948; Harley, 1990). Waterhyacinth infestations intensify mosquito problems by hindering insecticide application, interfering with predators, increasing habitat for species that attach to plants, and impeding runoff and water circulation (Seabrook, 1962).

Historically, annual losses associated with waterhyacinth-caused damages are often quite high, e.g., costs were estimated at $65 to 75 million in Louisiana during the 1940s (Gowanloch and Bajkov, 1948). During that same period, fish and wildlife losses alone in the six southeastern states exceeded $4 million per year in 1947 (Tabita and Woods, 1962). Holm *et al.*(1969) ascribed losses of $43 million in 1956 to waterhyacinth infestations in Florida, Mississippi, Alabama, and Louisiana, and early estimates from the US Army Corps of Engineers estimated benefits from waterhyacinth control programs at nearly $14 million in 1965 (Gordon and Coulson, 1974). Florida spent more than $43 million during 1980 to 1991 to suppress waterhyacinth and waterlettuce (Schmitz *et al.*, 1993), and more recently, annual costs for waterhyacinth management range from $500,000 in California to $3 million in Florida (Mullin *et al.*, 2000). The largest infestations occur in Louisiana, where the Department of Fisheries herbicidally treats about 25,000 acres of waterhyacinth per year, mostly at boat ramps, at an annual cost of $2 million (R. Brassette, pers. comm.).

Biological control measures have become increasingly attractive to manage waterhyacinth problems. Since the the early 1970s, USDA has studied biological control of waterhyacinth using the weevils *Neochetina eichhorniae* Warner, *Neochetina bruchi* Hustache, and the pyralid moth *Niphograpta (=Sameodes) albiguttalis* (Warren). These three agents and the mite, *Orthogalumna terebrantis* Wallwork, are now widely used in biological control programs targeted on waterhyacinth (Table 3).

Worldwide, biological control programs against waterhyacinth have reported successes (Julien and Griffiths, 1998), with the *Neochetina* weevils being successful. Biological control, however, often is not sufficient to adquate management of the waterhyacinth problem, since biological control agents tend to be slow-acting, which may not be acceptable for a particular application and biological control may not be compatible with other management practices (Center *et al.*, 1999a). Explosive growth of waterhyacinth stimulated by high nutrient levels may also limit the success of biological control measures (Heard and Winteron, 2000). Additional biological control agents have been identified, but their effectiveness at controlling waterhyacinth is incompletely characterized.

Table 3. Arthropods potentially acting as biological control agents for waterhyacinth

Species	Field and Laboratory Host Plants	Attributes, Limitations, and Current Status of Research
Neochetina eichhorniae Warner (Col.: Curculionidae)	*E. crassipes*	In use in North America, Australia, Africa, and Asia (Julien and Griffiths, 1998)
Neochetina brunchi Hustache (Col.: Curculionidae)	*E. crassipes*	Ibid.
Niphograpta albiguttalis (Warren) (Lep.: Pyralidae)	*E. crassipes*	Ibid.
Orthogalumna terebrantis Wallwork (Acarina: Galumnidae)	*E. crassipes, E. azurea, Pontederia cordata, Reussia subovata*	Ibid.

References and Supplemental Bibliography

APG (Angiosperm Phylogeny Group). 1998. An ordinal classification for the families of flowering plants. *Annals of the Missouri Botanical Garden* 85: 531-553.

Anonymous. 2000. Lake Victoria: Against the odds. *Water Hyacinth News* 1: 3-7.

Baer, R. G. and P. C. Quimby, Jr. 1982. Some natural enemies of the native moth *Arzama densa* Walker on waterhyacinth. *Journal of the Georgia Entomological Society* 17: 321-327.

Barrett, S. C. H. 1977. Tristyly in *Eichhornia crassipes* (Mart.) Solms (water hyacinth). *Biotropica* 9: 230-238.

Barrett, S. C. H. 1980. Sexual reproduction in *Eichhornia crassipes* (water hyacinth). II. Seed production in natural populations. *Journal of Applied Ecology* 17: 113-124.

Bennett, F. D. 1967. Notes on the possibility of biological control of the water hyacinth *Eichhornia crassipes*. *Pest Articles and News Summaries*, Section C 13(4): 304-309.

Bennett, F. D. 1972. Survey and assessment of the natural enemies of water hyacinth, *Eichhornia crassipes*. *Pest Articles and News Summaries* 18(3): 310-311.

Bennett, F. D. 1976. Current status of investigations on biotic agents of water hyacinth *Eichhornia crassipes* and water fern *Salvinia molesta*. Unpublished Report Commonwealth Institute of Biological Control, Curepe, Trinidad.

Beshir, M. O. and F. D. Bennett. 1985. Biological control of waterhyacinth on the White Nile, Sudan, pp. 491-496. *In* Delfosse, E. S. (ed.). *Proceedings of the VI International Symposium on Biological Control of Weeds*. August19-25, 1984. Vancouver, Canada, Agriculture Canada, Ottawa, Canada

Bickel, D. J. 1986. *Thrypticus* and an allied new genus, *Corindia*, from Australia (Dipt.: Doliochopodidae). *Records of the Australian Museum* 38: 135-151.

Bock, J. H. 1968. The water hyacinth in California. *Madrono* 19(7): 281-283.

Brou, V. A. Undated. Status in Louisiana of the introduced moth *Sameodes albiguttalis* (Warren) (Lepidoptera: Pyralidae). *Southern Lepidopterists Newletter*.

Buckingham, G. R. and S. Passoa. 1985. Flight muscle and egg development in waterhyacinth weevils, pp. 497-510. *In* Delfosse, E. S. (ed.). *Proceedings VI International Symposium on Biological Control of Weeds*. August19-25, 1984. Vancouver, British Columbia. Agriculture Canada, Ottawa, Canada.

Center, T. D. 1976. The potential of *Arzama densa* (Lepidoptera: noctuidae) for control of waterhyacinth with special reference to the ecology of waterhyacinth (*Eichhornia crassipes* (Mart.) Solms). Ph.D. dissertation, University of Florida, Gainesville, Florida, USA.

Center, T. D. 1982. The waterhyacinth weevils. *Neochetina eichhorniae* and *N. bruchi*. *Aquatics* 4(2): 8, 16, 18-19.

Center, T. D. 1984. Dispersal and variation in infestation intensities of waterhyacinth moth, *Sameodes albiguttalis* (Lepidoptera:Pyralidae) populations in peninsular Florida. *Environmental Entomolology* 13: 482-491.

Center, T. D. 1987a. Do waterhyacinth leaf age and ontogeny affect intra-plant dispersion of *Neochetina eichhorniae* (Coleoptera: Curculionidae) *eggs* and larvae? *Environmental Entomology* 16: 699-707.

Center, T. D. 1987b. Insects, mites, and plant pathogens as agents of waterhyacinth (*Eichhornia crassipes* [Mart.] Solms) leaf and ramet mortality. *Journal of Lake and Reservoir Managment*. 3: 285-293.

Center, T. D. 1994. Biological control of weeds: Waterhyacinth and waterlettuce, pp. 481-521. *In* Rosen, D., F. D. Bennett, and J. L. Capinera. (eds.). *Pest Management in the Subtropics: Biological Control - A Florida Perspective*. Intercept Publishing Company, Andover, United Kingdom.

Center, T. D. and F. A. Dray. 1992. Associations between waterhyacinth weevils (*Neochetina eichhorniae* and *N. bruchi*) and phenological stages of *Eichhornia crassipes* in southern Florida. *Florida Entomologist* 75: 196-211.

Center, T. D., and W. C. Durden. 1986. Variation in waterhyacinth/weevil interactions resulting from temporal differences in weed control efforts. *Journal of Aquatic Plant Management* 24: 28-38.

Center, T. D. and M. P. Hill. 1999. Host specificity of the pickerelweed borer, *Bellura densa* Walker (Lepidoptera: noctuidae) a potentially damaging natural enemy of water hyacinth, p. 67. *In* Hill, M. P., M. H. Julien, and T. D. Center (eds.). *Proceedings of the First Global Working Group Meeting for the Biological and Integrated Control of WaterHyacinth*, November 16-19, 1998, Harare, Zimbabwe. Plant Protection Research Institute, Pretoria, South Africa.

Center, T. D. and N. R. Spencer. 1981. The phenology and growth of waterhyacinth (*Eichhornia crassipes* (Mart.) Solms) in a eutrophic north-central Florida lake. *Aquatic Botany* 10: 1-32.

Center, T. D. and T. K. Van. 1989. Alteration of water hyacinth (*Eichhornia crassipes* [Mart.] Solms) leaf dynamics and phyto Chemistry by insect damage and plant density. *Aquatic Botany* 35: 181-195.

Center, T. D., J. K. Balciunas, and D. H. Habeck. 1982a. Identification and descriptions of

Sameodes albiguttalis (Warren) life stages. *Annals of the Entomological Society of America* 75: 471-479.

Center, T. D., K. K. Steward, and M. C. Bruner. 1982b. Control of waterhyacinth (*Eichhornia crassipes*) with *Neochetina eichhorniae* (Coleoptera: Curculionidae) and growth retardant. *Weed Science* 30: 453-457.

Center, T. D., A. F. Cofrancesco, and J. K. Balciunas. 1990. Biological control of wetland and aquatic weeds in the southeastern United States, pp. 239-262. *In* Delfosse (ed.). *Proceedings of the VII International Symposium on Biological Control of Weeds*, March 6-11, 1988, Rome, Italy. Commonwealth Scientific and Industrial Research Organization Publications, Melbourne, Australia.

Center, T. D., F. A. Dray, G. P. Jubinsky, and M. J. Grodowitz. 1999a. Biological control of waterhyacinth under conditions of maintenance management: can herbicides and insects be integrated? *Environmental Management* 23: 241-256.

Center, T. D., F. A. Dray, G. P. Jubinsky, and A. J. Leslie. 1999b. Waterhyacinth weevils (*Neochetina eichhorniae* and *N. bruchi*) inhibit waterhyacinth (*Eichhornia crassipes*) colony development. *Biological Control* 15: 39-50.

Charudattan, R., S. B. Linda, M. Kluepfel, and Y. A. Osman. 1985. Bio Control efficacy of *Cercospora rodmanii* on waterhyacinth. *Phytopathology* 75: 1263-1269.

Cock, M., R. Day, H. Herren, M. Hill, M. Julien, P. Neuenschwander, and J. Ogwang. 2000. Harvesters get that sinking feeling. *Biocontrol News and Information* 21(1): 1N-8N.

Cofrancesco, Jr., A. F. 1984. Biological control activities in Texas and California, pp. 57-61. *Proceedings of the 18th Annual Meeting of the Aquatic Plant Control Research Program*, November 14-17, 1983, Raleigh, North Carolina. Waterways Experiment Station Miscellaneous Paper A-84-4. U.S. Army Corps of Engineers, Vicksburg, Mississippi, USA.

Cofrancesco, Jr., A. F. 1985. Biological control of waterhyacinth and alligatorweed in Galveston District and at Jean Lafitte Park, Louisiana, pp. 103-109. *Proceedings of the 19th Annual Meeting of the Aquatic Plant Control Research Program*, November 26-29, 1984, Galveston, Texas. Waterways Experiment Station Miscellaneous Paper A-85-4, U.S. Army Corps of Engineers, Vicksburg, Mississippi, USA.

Cofrancesco, A. F., R. M. Stewart, and D. R. Sanders. 1985. The impact of *Neochetina eichhorniae* (Coleoptera: Curculionidae) on waterhyacinth in Louisiana, pp. 525-535. *In* Defosse, E. S. (ed.). *Proceedings of the VI International Symposium on Biological Control of Weeds*. August 19-25, 1984, Vancouver, Canada. Agriculture Canada, Ottawa, Ontario, Canada.

Cordo, H. A. 1999. New agents for biological control of water hyacinth, pp. 68-74. *In* Hill, M. P., M. H. Julien, and T. D. Center (eds.). *Proceedings of the First Global Working Group Meeting for the Biological and Integrated Control of WaterHyacinth*. November 16-19, 1998, Harare, Zimbabwe. Plant Protection Research Institute, Pretoria, South Africa.

Cordo, H. A. and C. J. DeLoach. 1975. Ovipositional specificity and feeding habits of the waterhyacinth mite, *Orthogalumna terebrantis*, in Argentina. *Environmental Entomology* 4: 561-565.

Cordo, H. A. and C. J. DeLoach. 1976. Biology of the waterhyacinth mite in Argentina. *Weed Science* 24: 245-249.

Cronquist, A. 1988. *The Evolution and Classification of Flowering Plants*, 2nd edition. New York Botanical Garden, New York.

Cruttwell, R. E. 1973. Preliminary investigations on some insects causing minor damage to waterhyacinth, *Eichhornia crassipes*. Report of the West Indian Station, Commonwealth Institute of Biological Control, Trinidad.

Delfosse, E. S. 1978. Effect on waterhyacinth of *Neochetina eichhorniae* [Col.: Curculionidae] combined with *Orthogalumna terebrantis* [Acari: Galumnidae]. *Entomophaga* 23: 379-387.

Delfosse, E. S., D. L. Sutton, and B. D. Perkins. 1976. Combination of the mottled waterhyacinth weevil and the white amur for biological control of waterhyacinth. *Journal of Aquatic Plant Management* 14: 64-67.

DeLoach, C. J. 1975. Identification and biological notes on the species of *Neochetina* that attack

Pontederiaceae in Argentina. (Coleoptera: Curculionidae:Bagoini). *Coleopterist Bulletin* 29(4): 257-265.

DeLoach, C. J., and H. A. Cordo. 1976a. Life cycle and biology of *Neochetina bruchi* and *N. eichhorniae*. *Annals of the Entomological Society of America* 69: 643-652.

DeLoach, C. J., and H. A. Cordo. 1976b. Ecological studies of *Neochetina bruchi* and *N. eichhorniae* on waterhyacinth in Argentina. *Journal of Aquatic Plant Management* 14: 53-59.

DeLoach, C. J., and H. A. Cordo. 1983. Control of waterhyacinth by *Neochetina bruchi* (Coleoptera: Curculionidae: Bagoini) in Argentina. *Environmental Entomology* 12: 19-23.

DeLoach, C. J., H. A. Cordo, R. Ferrer, and J. Runnacles. 1980. *Acigona infusellus*, a potential biological control agent for waterhyacinth: observations in Argentina (with descriptions of two new species of *Apanteles* by L. De Santis). *Annals of the Entomological Society of America* 73: 138-146.

De Quattro, J. 2000. Watch out water-hyacinth. *Agricultural Research* 48(3): 10-12.

Ding, J., R. Wang, W. Fu, and G. Zhang. 2001. Water hyacinth in China: Its distribution, problems and control status, pp. 29-32. *In* Julien, M., M. Hill, T. Center, and Ding, J. *Proceedings of the Meeting of the Global Working Group for the Biological and Integrated Control of Water Hyacinth*, Beijing, China, 9-12 October 2000. Australian Centre for International Agricultural Research, Canberra, Australia.

Dyte, C. E. 1993. The occurrence of *Thrypticus smaragdinus* Gest. (Dipt.: Dolio Chopodidae) in Britain, with remarks on plant hosts in the genus. *The Entomologist* 112: 81-84.

Eckenwalder, J. E. and S. C. H. Barrett. 1986. Phylogenetic systematics of the Pontederiaceae. *Systematic Botany* 11(3): 373-391.

Forno, I. W. 1983. Life history and biology of a waterhyacinth moth, *Argyractis subornata* (Lepidoptera: Pyralidae, Nymphulinae). *Annals of the Entomological Society of America* 76: 624-627.

Freeman, T. E. and R. Charudattan. 1984. *Cercospora rodmanii* Conway. A Biocontrol Agent for Waterhyacinth. Bulletin 842, Agricultural Experiment Stations, Institute of Food and Agricultural Sciences, University of Florida, Gainesville, Florida, USA.

Girling, D. J. 1983. Water hyacinth in the Sudan - early results. *Biological Control News and Information* 4(3):1.

Gopal, B. 1987. *Water Hyacinth*. Elsevier, New York.

Gordon, R. D. and J. R. Coulson. 1974. *Field observations of arthropods on waterhyacinth*. Aquatic Plant Control Program Technical Report 6:B3-B37, U.S. Army Corps of Engineers, Waterways Experiment Station, Vicksburg, Mississippi, USA.

Gowanloch, J. N. 1944. The economic status of the waterhyacinth in Louisiana. *Louisiana Conservationist* 2: 3-8.

Gowanloch, J. N. and A. D. Bajkov. 1948. Water hyacinth program. *Louisiana Department of Wildlife and Fisheries, Biennial Report* (1946/1947) 2: 66-124.

Goyer, R. A. and J. D. Stark. 1981. Suppressing water hyacinth with an imported weevil. *Ornamentals South* 3(6): 21-11.

Goyer, R. A. and J. D. Stark. 1984. The impact of *Neochetina eichhorniae* on waterhyacinth in southern Louisiana. *Journal of Aquatic Plant Management* 22: 57-61.

Habeck, D. H. and D. M. Lott. 1993 (unpublished). Indentification of immature stages of weevils associated with aquatic plants, pp. 279- 362. Final Report, USDA/ARS – IFAS/University of Florida Cooperative Agreement No. 58-43YK-9-0001, Integrated Management of Aquatic Weeds, October 1, 1988 to September 30, 1993.

Hahn, W. J. 1997. Commelinanae. http://phylogeny.arizona.edu/tree/...ons/commelinanae/ commelinanae.html (accessed 8 Aug. 2001).

Haigh, J. C. 1936. Notes on the water hyacinth (*Eichhornia crassipes* Solms) in Ceylon. *Ceylon Journal of Science* (A). 12(2): 97-108.

Haller, W. T., and D. L. Sutton. 1973. Effects of pH and high phosphorus concentrations on growth of waterhyacinth. *Hyacinth Control Journal* 11: 59-61.

Hansen, K. L., E. G. Ruby, and R. L. Thompson. 1971. Trophic relationships in the water hyacinth community. *Quarterly Journal of the Florida Academy of Science* 34(2): 107-113.

Harley, K. L. S. 1990. The role of biological control in the management of water hyacinth, *Eichhornia crassipes*. *Bio Control News and Information* 11(1): 11-22.

Heard, T. A. and S. L. Winterton. 2000. Interactions between nutrient status and weevil herbivory in the biological control of water hyacinth. *Journal of Applied Ecology* 37: 117-127.

Hill, M. P. and C. J. Cilliers. 1999. A review of the arthropod natural enemies,and factors that influence their efficacy, in the biological control of waterhyacinth, *Eichhornia crassipes* (Mart.) Solms-Laubach (Pontederiaceae), in South Africa. *African Entomology Memoir No. 1*: 103-112.

Hill, M. P. and T. Olckers. 2001. Biological control initiatives against water hyacinth in South Africa: Constraining factors, success and new courses of action, pp.33-38. *In* Julien, M., M. Hill, T. Center, and Ding, J. *Proceedings of the Meeting of the Global Working Group for the Biological and Integrated Control of Water Hyacinth*, Beijing, China, 9-12 October 2000. Australian Centre for International Agricultural Research, Canberra, Australia.

Hill, M. P., C. J. Cilliers, and S. Neser. 1999. Life history and laboratory host range of *Eccritotarsus catainensis* (Carvalho) (Heteroptera: Miridae), a new natural enemy released on waterhyacinth (*Eichhornia crassipes* (Mart.)Solms-Laub) (Pontederiaceae) in South Africa. *Biological Control* 14:127-133.

Hill, M. P., T. D. Center, J. N. Stanley, H. A. Cordo, J. Coetzee, and M. Byrne. 2000. The performance of the water hyacinth mirid, *Eccritotarsus catarinensis*, on water hyacinth and pickerelweed: a comparison of laboratory and field results, pp. 357-366. In Spencer, N. R. (ed.). *Proceedings of the Xth International Symposium on the Biological Control of Weeds*, Bozeman, Montana, USA, July 4-9, 1999. Montana State University, Bozeman, Montana.

Hitchcock, A. E., P. W. Zimmerman, H. Kirkpatrick, Jr., and T. T. Earle. 1950. Growth and reproduction of water hyacinth and alligator weed and their control by means of 2,4-D.

Contributions of the Boyce Thompson Institute 16(3): 91-130.

Holm, L. G., L. W. Weldon, and R. D. Blackburn. 1969. Aquatic weeds. *Science* 166: 699-709.

Holm, L. G., D. L. Plucknett, J. V. Pancho, and J. P. Herberger. 1977. *The World's Worst Weeds: Distribution and Biology*. University Press, Honolulu, Hawaii.

Jayanth, K. P. 1987. Suppression of water hyacinth by the exotic insect *Neochetinaeichhorniae* in Bangalore, India. *Current Science* 56(10):494-495.

Jayanth, K. P. 1988. Biological control of water hyacinth in India by release of the exotic weevil *Neochetina bruchi*. *Current Science* 57(17): 968-970.

Jones, R., and C. J. Cilliers. 1999. Integrated control of water hyacinth on the Nseleni/Mposa rivers and Lake Nsezi in KwaZulu-Natal, South Africa, pp.160-167. In Hill, M. P., M. H. Julien, and T. D. Center (eds.). *Proceedings of the First IOBC Global Working Group Meeting for the Biological and Integrated Control of Water Hyacinth*. November 16-19, 1998, Harare, Zimbabwe. Plant Protection Research Institute, Pretoria,South Africa.

Julien, M. 2001. Biological control of water hyacinth with arthropods: a review to 2000, pp.8-20. *In* Julien, M., M. Hill, T. Center, and Ding, J. *Proceedings of the Meeting of the Global Working Group for the Biological and Integrated; Control of Water Hyacinth*, Beijing, China, 9-12 October 2000. Australian Centre for International Agricultural Research, Canberra, Australia.

Julien, M. H. and M. W. Griffiths. 1998. Biological Control of Weeds. A World Catalogue of Agents and their Target Weeds, 4th edition. CABI Publishing, New York.

Julien, M. H. and W. Orapa. 1999. Structure and management of a successful biological control project for water hyacinth, pp. 123-134. *In* Hill, M. P., M.H Julien, and T.D. Center (eds.) *Proceedings of the 1st IOBC Global Working GroupMeeting for the Biological and Integrated Control of Water Hyacinth*.November 16-19, 1998, Harare, Zimbabwe. Plant Protection Research Institute, Pretoria, South Africa.

Julien, M. H., and J. Stanley. 1999. Recent research on biological control for water hyacinth in Australia, pp. 52-61. *In* Hill, M. P., M.H Julien, and T. D. Center (eds.) *Proceedings of the 1st*

IOBC Global Working Group Meetingfor the Biological and Integrated Control of Water Hyacinth. November16-19, 1998, Harare, Zimbabwe. Plant Protection Research Institute, Pretoria, South Africa.

Julien, M. H., M. W. Griffiths, and A. D. Wright. 1999. *Biological Control of Water Hyacinth*. ACIAR, Canberra, Australia. 87 pp.

Kiefer, H. H. 1979. *Eriophyid studies* C-17. Science and Education Adminstration, Agricultural Research Service, U.S. Dep. of Agriculture, Issued August 27, 1979, 24 pp.

Knipling, E. B., S. H. West, and W. T. Haller. 1970. Growth characteristics, yield potential, and nutritive content of water hyacinths. *Proceedings of the Soil and Crop Science Society of Florida* 30: 51-63.

Kohji, J., R. Yamamoto, and Y. Masuda. 1995. Gravitropic response. I. *Eichhorniacressipes* [sic] (water hyacinth). I. Process of gravitropic bending in the peduncle. *Journal of Plant Research* 108: 387-393.

Labrada, R. 1996. Status of water hyacinth in developing countries, pp. 3-11. *In* Charudattan, R., R. Labrada, T. D. Center, and C. Kelly-Begazo (eds.).*Strategies for Water Hyacinth Control*. Report of a Panel of Experts Meeting, September 11-14, 1995, Fort Lauderdale, Florida, USA. FAO, Rome, Italy.

Manning, J. H. 1979. Establishment of waterhyacinth weevil populations in Louisiana. *Journal of Aquatic Plant Management* 17: 39-41.

Matthews, L. J. 1967. Seedling establishment of water hyacinth. *Pest Articles and News Summaries* (C) 13(1): 7-8.

McVea, C. and C. E. Boyd. 1975. Effects of waterhyacinth cover on water chemistry, phytoplankton, and fish in ponds. *Journal of Envirnomental Quality* 4(3): 375-378.

Mitchell, D. S. and P. A. Thomas. 1972. *Ecology of Water Weeds in the Neotropics*. UNESCO, Paris, France.

Morris, M.J. 1990. *Cercospora piaropi* recorded on the aquatic weed, *Eichhornia crassipes*, in South Africa. *Phytophylactica* 22: 255-256.

Morris, M. J., A. R. Wood, and A. den BreeÃ¿en. 1999. Plant pathogens and biological control of weeds in South Africa: a review of projects and progress during the last decade. *African Entomology Memoir* 1: 129-137.

Mullin, B. H., L. W. J. Anderson, J. M. DiTomaso, R. E. Eplee, and K. D. Getsinger. 2000. Invasive plant species. Issue Paper No. 13. Council for Agricultural Science and Technology, Ames, Iowa.

Muramoto, S., I. Aoyama, and Y. Oki. 1991. Effect of salinity on the concentration of some elements in water hyacinth (*Eichhornia crassipes*) at critical levels. *Journal of Environmental Science and Health* A26(2): 205-215.

O'Brien, C. W. 1976. A taxonomic revision of the new world subaquatic genus *Neochetina*. *Annals of the Entomological Society of America* 69(2): 165-174.

Ogwang, J. A. and R. Molo. 1999. Impact studies on *Neochetina bruchi* and *Neochetinaeichhorniae* in Lake Kyoga, Uganda, pp. 10-13. In Hill, M. P., M. H. Julien, and T. D. Center (eds.). *Proceedings of the 1st IOBC Global Working Group Meeting for the Biological and Integrated Control of Water Hyacinth*. November 16-19, 1998, Harare, Zimbabwe. Plant Protection Research Institute, Pretoria, South Africa.

O'Hara, J. 1967. Invertebrates found in water hyacinth mats. *Quarterly Journal of the Florida Academy of Science* 30(1): 73-80.

Parija, P. 1934. Physiological investigations on water-hyacinth (*Eichhornia crassipes*)in Orissa with notes on some other aquatic weeds. *Indian Journal of Agricultural Science* 4: 399-429.

Penfound, W. T. and T. T. Earle. 1948. The biology of the water hyacinth. *Ecological Monographs* 18: 447-472.

Perkins, B. D. 1974. Arthropods that stress water hyacinth. *Pest Articles and News Summaries* 20(3): 304-314.

Richards, J. H. 1982. Developmental potential of axillary buds of water hyacinth, *Eichhornia crassipes* Solms. (Pontederiaceae). *American Journal of Botany* 69: 615-622.

Sabrosky, C. W. 1974. *Eugaurax setigena* (Diptera: Chloropidae), a new stem miner in water hyacinth. *Florida Entomologist* 57: 347-348.

Sands, D. P. A. and R. C. Kassulke. 1983. *Acigona infusellus* (Walker) (Lepidoptera:Pyralidae), an agent for biological control of waterhyacinth (*Eichhorniacrassipes*) in Australia. *Bulletin of Entomological Research* 73: 625-632.

Schmitz, D. C., J. D. Schardt, A. J. Leslie, F. A. Dray, Jr., J. A. Osborne, and B. V.Nelson. 1993. The ecological impact and management history of three invasive alien plant species in Florida, pp. 173-194. *In* McKnight,B. N. (ed.).*Biological Pollution*. *The Control and Impact of Invasive Exotic Species*. Indiana Academy of Science, Indianapolis, Indiana, USA.

Seabrook, E. L. 1962. The correlation of mosquito breeding to hyacinth plants. *Hyacinth Control Journal* 1: 18-19.

Silveira Guido, A. 1965 (Unpublished report). Natural enemies of weed plants. Final report on PL-480 Project S9-CR-1 (Jan. 1962 to 15 Nov. 1965). Universidad de la Republica, Facultad de Agronomia, Montevideo, Uruguay.

Silveira Guido, A. 1971. Datos preliminaries de biologia y especificidad de *Acigona ignitalis* Hamps. (Lep., Pyralidae) sobre el hospedero *Eichhorni acrassipes* (Mart.) Solms-Laubach (Pontederiaceae). *Revista de la So Ciedad de Entomologie de Argentina* 33(1-4): 137-145.

Silveira Guido, A. and B. D. Perkins. 1975. Biology and host specificity of *Cornops aquaticum* (Bruner) (Orthoptera: Acrididae), a potential biologicalcontrol agent for waterhyacinth. *Environmental Entomology* 4: 400-404.

Stanley, J. N. and M. H. Julien. 1999. The host range of *Eccritotarsus catarinensis* (Heteroptera: Miridae), a potential agent for the biological control of waterhyacinth (*Eichhornia crassipes*). *Biological Control*14: 134-140.

Stewart, R. M. 1985. Biological control of waterhyacinth in the California Delta, pp. 110-112.

Proceedings of the 19th Annual Meeting of the Aquatic Plant Control Research Program, November 26-29, 1984, Galveston, Texas. Waterways Experiment Station Miscellaneous Paper A-85-4, U.S. Army Corps of Engineers, Vicksburg, Mississippi, USA.

Stewart, R. M. 1987. Dispersing waterhyacinth bio Control agents in the Galveston District, pp. 105-107. *Proceedings of the 21th Annual Meeting of the Aquatic Plant Control Research Program*. November 17-21, 1986, Mobile, Alabama. Waterways Experiment Station Miscellaneous Paper A-87-2, U.S. Army Corps of Engineers, Vicksburg, Mississippi, USA.

Tabita, A. and J. W. Woods. 1962. History of hyacinth control in Florida. *Hyacinth Control Journal* 1: 19-23.

Takhtajan, A. 1997. *Diversity and Classification of Flowering Plants*. Columbia University Press, New York.

Tharp, B. C. 1917. Texas parasitic fungi – new species and amended descriptions *Mycologia* 9: 105-124.

Thorne, R. F. 1992. Classification and geography of flowering plants. *Botanical Review* 58: 225-348.

Ueckermann, C. and Hill, M.P. 2001. Impact of herbicides used in water hyacinth control on the natural enemies released against the weed for biological control. Report to the Water Research Commission. WRC Report No. 915/1/01 (July 2001). Water Research Commission, Pretoria, South Africa. 81 pp.

Ueki, K. 1978. Habitat and nutrition of waterhyacinth. *Japan Agricultural Research Quarterly* 12(3): 121-127.

Ultsch, G. R. 1973. The effects of waterhyacinth (*Eichhornia crassipes*) on the microenvironment of aquatic communities. *Archiv fuer Hydrobiologie* 72: 460-473.

USDA, NRCS (U.S. Department of Agriculture, Natural Resources Conservation Service). 1999. The PLANTS Database (http://plants.usda.gov/plants). (11 Aug. 2001)

Vogel, E. and A. D. Oliver, Jr. 1969a. Evaluation of *Arzama densa* as an aid in the control of waterhyacinth in Louisiana. *Journal of Economic Entomology* 62(1): 142-145.

Vogel, E. and A. D. Oliver, Jr. 1969b. Life history and some factors affecting the population of *Arzama densa* in Louisiana. *Annals of Entomological Society of America* 62: 749-752.

Warner, R. E. 1970. *Neochetina eichhorniae*, a new species of weevil from waterhyacinth, and biological notes on it and *N. bruchi* (Coleoptera: Curculionidae: Bagoini). *Proceedings of the EntomologicalSociety of Washington* 72: 487-496.

Watson, M. A. 1984. Developmental constraints: effect on population growth and patterns of resource allo Cation in a clonal plant. *American Naturalist* 123: 411-426.

Watson, M. A., and Cook, C. S. 1982. The development of spatial pattern in clones of an aquatic plant, *Eichhornia crassipes* Solms. *American Journal of Botany* 69: 248-253.

Watson, M. A., and Cook, G. S. 1987. Demographic and developmental differences among clones of water hyacinth. *Journal of Ecology* 75: 439-457.

Webber, H. J. 1897. *The water hyacinth, and its relation to navigation in Florida*. Bulletin No. 18. U.S. Department of Agriculture, Division of Botany. Washington, D.C.

Weber, H. 1950. Morphologische und anatomische studien Ã¼ber *Eichhornia crassipes* (Mart.) Solms. *Adhandllungen der Mathematisch - Naturwissenschattlicher Klasse, Akademie der Wissenhaften und der Literature Mainz* 6: 135-161.

Weed Science Society of America (WSSA). 1984. Composite list of weeds. *Weed Science* 32 (Suppl. 2): 1-137.

Wright, A. D. 1979. Preliminary report on damage to *Eichhornia crassipes* by an introduced weevil at a central Queensland liberation site, pp. 227-229.*In* Medd, R. W. and B. A. Auld (eds). *Proceedings of the 7th Asian-Pacific Weed Science So Ciety Conference*, November 26-30, 1979, Sydney, Australia. Council of Australian Weed Science So Cieties for the Asian-Pacific Weed Science Society.

Wright, A. D. 1981. Biological control of waterhyacinth in Australia, pp. 529-535. *In* Delfosse, E. S. (ed.). *Proceedings of the V International Symposium onBiological Control of Weeds*. Brisbane, Australia, July, 1980. Commonwealth Scientific and Industrial Research Organization Publications, Melbourne, Australia.

Wright, A. D., and T. D. Center. 1984. Predicting population intensity of adult*Neochetina eichhorniae* (Coleoptera: Curculionidae) fromincidence of feeding on leaves of waterhyacinth, *Eichhornia crassipes*. *Environmental Entomology* 13: 1478-1482.

Zeiger, C. F. 1962. Hyacinth – obstruction to navigation. *Hyacinth Control Journal* 1: 16-17.

[25]*Lythrum salicaria* L.

Common name: Purple loosestrife

Taxonomy and identification. Purple loosestrife is an erect rhizomatous perennial herb in the loosestrife family, with a square to angular, woody stem having opposite or whorled leaves. The plant's native range was in Eurasia and Africa, but its introduction outside its native area has yielded plant infestations at a wide range of moist or marshy sites throughout North America. Stems are erect (1.5 to 8 or more feet tall), four to six angled, and can be smooth or pubescent with few branches. Leaves are simple (0.75 to 4 inches long, 0.2 to 0.5 inches wide), entire, and can be opposite or whorled. Leaves are lance-shaped, stalkless, and heart-shaped or rounded at the base. Plants are usually covered by a downy pubescence. Loosestrife plants grow from four to ten feet high, depending upon conditions, and produce a showy display of magenta-colored flower spikes throughout much of the summer. Flowers have five to seven petals. Mature plants can have from 30 to 50 stems arising from a single rootstock.

Life history and biology. The most identifiable characteristic of purple loosestrife is the striking rose to purple colored flowers. The flowers are arranged on a spike, which can be a few inches to 3 feet long. Each flower has five to seven petals arising from a cylindrical green tube. At mid-continent latitudes characteristic of the northern Great Plains, plants usually flower from early July to mid-September. The seed capsule is two celled and contains many very small seeds (1 mm long or less). The roots become thick and woody in mature plants. The aerial shoots die in the fall and new shoots arise the following spring from buds at the top of the root crown. Although the root crown expands and produces more shoots each year, the maximum growth of the root crown diameter is limited to about 20 inches.

Spread of purple loosestrife is primarily by seed, but the plant can also spread vegetatively from stem cuttings. Seed viability varies from 50 to 100 percent, and approximately 2.7 million seeds are produced per plant, giving purple loosestrife the potential to spread rapidly once established in an area.

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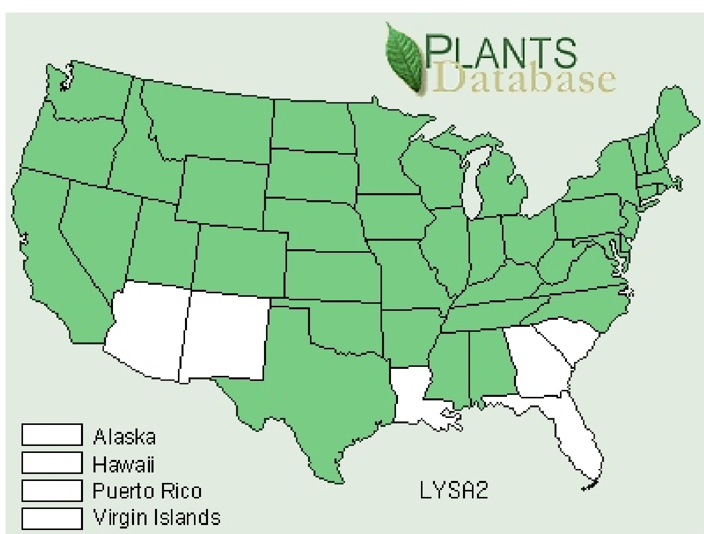
[25]Original material prepared by Jil M. Swearingen, U.S. National Park Service, Washington, DC; W-1132 (Revised) March 2002, Rodney G. Lym, Professor, Plant Sciences, North Dakota State University, NDSU Extension Service. Updated, edited, and revised November, 2004.

Purple loosestrife is capable of invading many wetland types, including freshwater wet meadows, tidal and non-tidal marshes, river and stream banks, pond edges, reservoirs, and ditches. Purple loosestrife has an extended flowering season, generally from June to September (depending on latitude and site-specific conditions), which allows it to produce vast quantities of seed. Flowers require pollination by insects, for which it supplies an abundant source of nectar. A mature plant may have as many as thirty flowering stems capable of producing an estimated two to three million, minute seeds per year. Purple loosestrife readily reproduces vegetatively through underground stems at a rate of about one foot per year. Many new stems may emerge vegetatively from a single rootstock of the previous year.

The most destructive impact of purple loosestrife invasions is adverse ecological effects associated with its dense monotypic stands as it displaces native wetland plants. Under optimum conditions, a small isolated group of purple loosestrife plants can spread to cover aquatic sites in just one growing season. When purple loosestrife replaces native vegetation, it also can displace wildlife. For example, songbirds do not consume the small hard seed. Muskrats use cattails to build their homes, and they show a preference for cattail over purple loosestrife for food. Waterfowl, especially ducks, avoid wetlands that have become dominated with purple loosestrife. In addition, overall waterfowl production decreases as suitable nesting habitat is eliminated. The plant's growth is generally too compact to offer cover, and cover may be as crucial to wildlife as food.

Native range. *Lythrum salicaria* has a native range throughou Eurasia, stretching from Great Britain, across central and southern Europe to central Russia, Japan, Manchuria China, then southeast Asia and northern India.



Non-indigenous range. According to the U.S. Fish and Wildlife Service and other survey records (e.g., BONAP, see Kartesz 1999), purple loosestrife now occurs in nearly every state of the US (Figure 27; states in green have records of local populations). The map in Figure 27 does not reflect relatively recent additions with the states of South Carolina, Georgia, Louisiana, New

Mexico, and Arizona. Currently, only Florida remains without a documented observation of purple loosestrife. Various states in the US and provinces in Canada have identified the species as a "noxious weed" or similar descriptor (Kartesz 1999). For example, North Dakota, Minnesota, and Manitoba each have programs dedicated to eradication of purple loosestrife, given the species current distribution within each's political boundaries (Figure 27 - Figure 29).

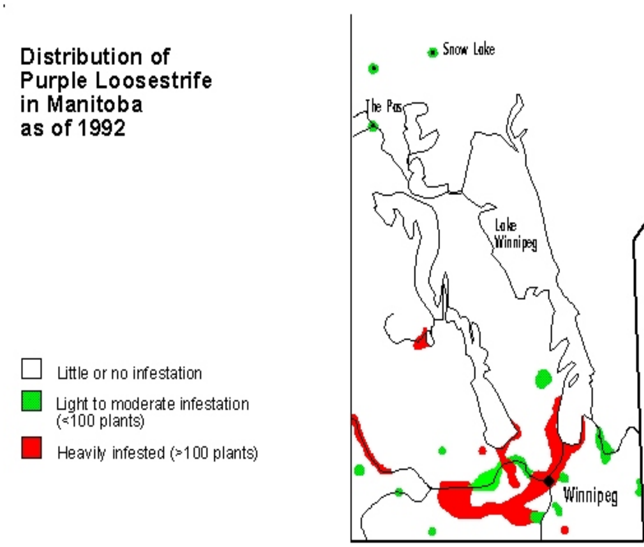


Figure 27. Purpleloosestrife in Manitoba.

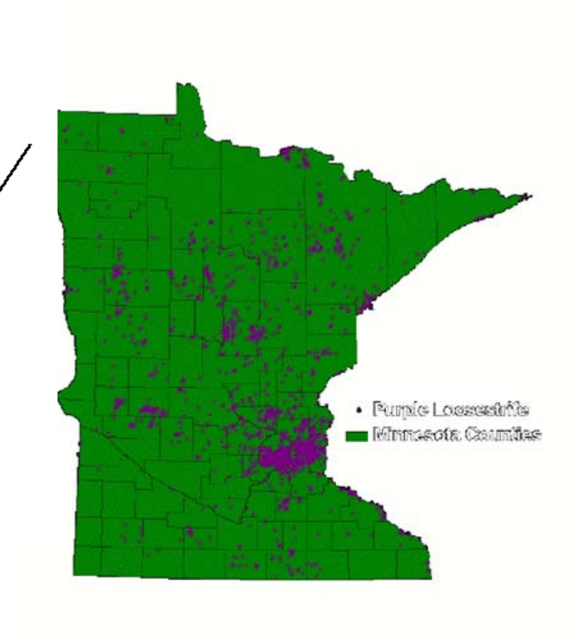


Figure 28. Purpleloosestrife in Minnesota.

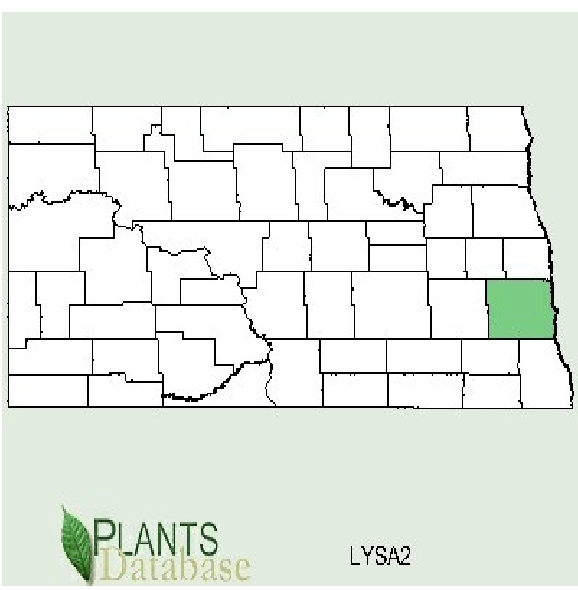


Figure 29. Purpleloosestrife in North Dakota (county record).

Impacts and control. Purple loosestrife was first introduced into North America in the early 1800's. Plants were sold throughout the US as various horticultural cultivars, all having striking colors and being easily grown perenniels (e.g., winter hardy and lack of insect or disease problems). These garden cultivars were thought to be sterile but have now been shown to cross-pollinate with the wild *Lythrum* type and sometimes with other *Lythrum* cultivars.

Purple loosestrife adapts readily to natural and disturbed wetlands. As it establishes and expands its local and regional range, the species out-competes and replaces native grasses, sedges, and other flowering plants that provide a higher quality source of nutrition for wildlife. The highly invasive nature of purple loosestrife allows it to form dense, homogeneous stands that restrict native wetland plant species, including some federally endangered orchids, and reduce habitat for waterfowl.

Several methods are available for purple loosestrife control, including mechanical, biological, and chemical. The size and location of a specific infestation will determine the best control methods. In general, small infestations of a few plants can be controlled by digging, especially when plants are only a few years old. Larger infestations require treatment with herbicides and/or biological control agents.

Mechanical control. Small infestations can be controlled by removing all roots and underground stems. It is difficult to remove all of the roots in a single digging, so monitor the area for several growing seasons to ensure that purple loosestrife has not regrown from roots or seed. This method is most useful on garden plantings or young infestations.

Dispose of plants and roots by drying and burning or by composting in an enclosed area. Take care to prevent further seed spread from clothing or equipment during the removal process. Removal of all plant material is important. Small segments of purple loosestrife stems can become rooted and reestablish the infestation.

Chemical control. Herbicides can be used to control purple loosestrife in areas too large to be controlled by digging. Also, herbicides can be applied to individual plants selectively in landscape situations to prevent killing desirable plants. Infestations growing along streams or in marshy areas may require specialized equipment and application by trained professionals.

Glyphosate (Rodeo® or Roundup®, various other trade names for glyphosate) will provide good control of purple loosestrife when applied from July to early September. Many formulations of glyphosate are sold but only those labeled for aquatic use can be applied in or near water. For example, the Rodeo and Glypro formulations of glyphosate can be used in water. With the Rodeo or Glypro formulations, a nonionic surfactant approved for aquatic sites at 0.25% vol/vol must be added to the spray solution. Roundup and similar glyphosate formulations can be used to remove purple loosestrife from large plantings or infestations away from water. Glyphosate has no soil residual so it could be used to remove purple loosestrife located within an ornamental planting without having to dig in the flower bed. Best results have been obtained when glyphosate is applied as a 1 to 1.5% concentration (1 to 1.5 gallons glyphosate per 100 gallons of water) or (1.3 to 1.9 fl. oz./gallon of water) at bloom or shortly thereafter.

A variety of sprayers, including backpack sprayers and boat-mounted sprayers, can be used to control purple loosestrife in aquatic sites. Wick application is also effective but is labor intensive. Spray dye added to the tank may be useful to ensure uniform application to purple loosestrife with minimal herbicide applied to desirable plants.

Eliminating the entire vegetative cover will promote purple loosestrife seed germination, which can result in an increase in plant density rather than control. Since glyphosate does not provide residual control, treated areas will need to be monitored for regrowth from the roots or seedlings for several years. A 2,4-D formulation labeled for use near water applied as a 2% solution (2 gallons 2,4-D per 100 gallons of water) or (2.6 fl. oz./gallon of water) will prevent seedling establishment when applied in early fall or spring before the plants can establish perennial characteristics.

Triclopyr (trade name Garlon®) is a selective broadleaf herbicide that will not kill cattail or other desirable monocot species. However, Garlon is not labeled for use in water, and it can only be used up to the water's edge. Garlon will provide good to excellent purple loosestrife control when applied in the pre to early flower or late flower growth stages. Garlon should be applied as a 1 to 2% solution (1 to 2 gallons Garlon® per 100 gallons of water or 1.3 to 2.6 fl. oz./gallon of water) and will provide some residual seedling control. Garlon® can be applied in dryland sites but should not be used in landscapes or flower beds because soil residual of the herbicide may prevent establishment of other horticultural plants.

Regardless of the herbicide applied, the infested areas should be monitored to ensure that purple loosestrife does not reinfest from root or seed. Also, areas downstream from river or creek infestations and on all sides of a lake or pond infestation should be monitored for purple loosestrife seedlings.

Biological control. While herbicides and hand removal may be useful for controlling individual plants or small populations, biological control is seen as the most likely candidate for effective long term control of large infestations of purple loosestrife. As of 1997, three insect species from Europe have been approved by the US Department of Agriculture for use as biological control agents. These plant-eating insects include a root-mining weevil (*Hylobius transversovittatus*), and two leaf-feeding beetles (*Galerucella calmariensis* and *Galerucella pusilla*). Two flower-feeding beetles (*Nanophyes*) that feed on various parts of purple loosestrife plants are still under investigation. *Galerucella* and *Hylobius* have been released experimentally in natural areas in 16 northern states, from Oregon to New York. Although these beetles have been observed occasionally feeding on native plant species, their potential impact to non-target species is considered to be low. Of these insects, the two *Galerucella* spp. leaf feeding beetles have been most successful. For example, in North Dakota these insects overwinter as adults and lay eggs in early June in North Dakota. The adults and especially the larvae feed on the leaves and flowers of purple loosestrife. Following several summers of heavy feeding, purple loosestrife infestations have been greatly reduced. However, since the largest infestations in North Dakota are in urban areas, mosquito control programs have kept these insects from becoming well established. Purple loosestrife infestations in North Dakota are generally small and isolated and should be controlled by chemical and/or mechanical methods.

Regardless of the control methods employed, successful weed management must limit future dispersal, and locally or regionally this may be accomplished using chemical controls such as herbicides, physical means such as mechanical removals, and biological controls. Depending upon the size and location of the outbreak to be control, a combination of these management tools may be applied to prevent or control spread of purple loosestrife. For small infestations of young purple loosestrife, mechanical removal, e.g., plants may be pulled by hand, should be completd before seed set. For older plants, spot treatments with glyphosate herbicide (e.g., Rodeo® for wetlands, Roundup® for uplands) is recommended. These herbicides may be most effective when applied late in the season when plant are preparing for dormancy. However, it may be best to do a mid-summer and a late season treatment, to reduce the amount of seed produced.

References

Heidorn, R. and B. Anderson. 1991. Vegetation management guideline: purple loosestrife (*Lythrum salicaria* L.). Natural Areas Journal 11:172-173.

LaFleur, A. 1996. Invasive plant information sheet: purple loosestrife. The Nature Conservancy, Connecticut Chapter.

Malecki, R.A. (et al.). 1993. Biological control of purple loosestrife (*Lythrum salicaria*). BioScience 43 (10):680-686.

The Nature Conservancy. <u>Purple Loosestrife: Element Stewardship Abstract</u> . In: Wildland Weeds Management & Research Program, Weeds on the Web.

Thompson, Daniel Q., Ronald L. Stuckey, Edith B. Thompson. 1987. Spread, Impact, and Control of Purple Loosestrife (*Lythrum salicaria*) in North American Wetlands . U.S. Fish and Wildlife Service. 55 pages.

Virginia Department of Conservation and Natural Resources. 1995. Invasive Alien Plant Species of Virginia: Purple Loosestrife (*Lythrum salicaria*).

[26]***Tamarix* spp.** (most likely species of concern include *Tamarix ramosissima* Ledebour, *Tamarix pentandra* Pallas, *Tamarix chinensis* Loureiro, *Tamarix parviflora* De Candolle and hybrids)

Common names: Saltcedar, Tamarisk

Taxonomy and identification. Commonly referred to as saltcedar or tamarisk, the plant is a member of the Tamaricaceae, and not unlike other invasive species issues, the plant's taxonomy clouds management issues as that relates to prevention and control of the plant's continued spread throughout North Ameria. For example, Robinson (1965) stated that two species of *Tamarix* have escaped cultivation in western North America, namely *T. pentandra* Pallas and *T. gallica* L., yet Horton and Campbell (1974) studied tamarisk collections from the southwestern United States and grew plants under controlled conditions, then proposed assigning all deciduous life-forms to a single species, *T. chinensis*. Yet another study (Welsh et al. 1987) classified deciduous tamarisk species in Utah as either *T. ramosissima* which has flower parts in 5's (5-merous) or *T. parviflora* which has flower parts in 4's (4-merous). According to Weber (1990), *T. ramosissima* Ledebour and *T. chinensis* Loureiro were taxonomic synonyms, and Sudbrock (1993) stated that *T. ramosissima* and *T. chinensis* were difficult to distinguish, appeared to hybridize, and may be lumped together as *T. chinensis*. Other researchers lump all deciduous tamarisk species into *T. pentandra*. In practice, little distinction is made among the deciduous tamarisk species for management purposes, and Carpenter (2004) followed an increasingly common practice of referring to all 5-merous deciduous tamarisk species that have become naturalized in western North America as *T. ramosissima*, and the 4-merous deciduous species as *T. parviflora*. The 5-merous deciduous tamarisk appears to be more widespread in North America than the 4-merous species.

Deciduous tamarisk species in the western United States – *T. ramosissima* or *T. parviflora* – can be distinguished using the characteristics in Table 4. Both species are deciduous, loosely branched shrubs or small trees. The branchlets are slender with minute, appressed scaly leaves. The leaves are rhombic to ovate, sharply pointed to gradually tapering, and 0.5 –3.0 mm long. The margins of the leaves are thin, dry and membranaceous. White to pink flowers are most abundant between

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[26]Original material prepared by Alan T. Carpenter, Land Stewardship Consulting, Boulder CO, and maintained by The Nature Conservancy Wildland Weed Management and Research Program, University of California, Davis, CA (Ramona A. Robison and John M. Randall, The Nature Conservancy, Wildland Weeds Management and Research, 124 Robbins Hall, University of California, Davis, CA 95616). Updated January 2004.

April and August, but may be found any time of the year, and are borne on slender racemes 2-5 cm long on the current year's branches. Flowers are grouped together in terminal panicles borne on short pedicels. Petals are usually retained on the fruit, and seeds are borne in a lance-ovoid capsule 3-4 mm long. Seeds contain no endosperm, are about 0.45 mm long and 0.17 mm wide, and have unicellular hairs about 2 mm long at the apical end (Wilgus and Hamilton 1962; Stevens 1990).

Table 4. Distinguishing characteristics of *T. ramosissima* and *T. parviflora* based on Welsh et al. (1987).

Characteristic	***Tamarix ramosissima***	***Tamarix parviflora***
Size	< 5 m tall	< 6 m tall
Bark	reddish brown	dark brown to deep purple
Bracts	scarcely translucent	more or less translucent
Flowers	parts in 5s	parts in 4s
Sepals	outer two narrower than inner	outer two keeled and acute
	all more or less acute	outer flat or slightly keeled and obtuse
Stamen filaments	inserted under the disc near the margin	arising gradually from disc lobes between the lobes
Petals	obovate, 1-1.8 mm long	oblong to ovate, 1.9-2.3 mm long

Life history and biology. Throughout the western US, various species of *Tamarix* are increasingly a management problem, especially in riparian areas where the plant has a long history of successful invasions and a correspondingly long history of adverse impacts to the system. Tamarisk is an aggressive, woody invasive plant species that has become established over as much as a million acres of floodplains, riparian areas, wetlands and lake margins in the western United States (Johnson 1986). Once established, the plant is a relatively long-lived and can tolerate a wide range of environmental conditions. Massive quantities of small seeds are produced annually by each plant, and asexual propagation occurs from buried or submerged stems.

Tamarisk displaces and eventually replaces native woody species such as cottonwood, willow and mesquite, which occupy habitats similar to those preferred by tamarisk. When riparian habitats are altered, for example, by augmented stream flows (e.g., when timing and amount of peak water discharge, salinity, temperature, and substrate texture have been altered by human activities), tamarisk may be favored in competitive interactions with native species of riparian wood shrubs and trees. Stands of tamarisk generally have lower wildlife values compared to stands of native vegetation, although tamarisk can be important to some bird species as nesting habitat.

Tamarisk is a facultative phreatophyte, drawing on groundwater sources as available, but once established, access to groundwater is not required for its survival. Tamarisk translocates large quantities of water, possibly more than woody native plant species that occupy similar habitats. The species is a halophyte and can tolerant highly saline habitats, in part, by concentrating salts in its leaves (hence, the species common name). Over time, as leaf litter accumulates under tamarisk plants, the surface soil can become highly saline, thus impeding future colonization by many native plant species.

Although it grows mostly on fine-textured soils (Everitt 1980), tamarisk can grow in many different substrates from below sea level to about 2100 m elevation (Hoddenbach 1990). As a facultative phreatophyte, tamarisk occurs in areas where its roots can reach the water table, such as floodplains, along irrigation ditches and on lake shores. Plants usually grow where the depth to ground water does not exceed 3 to 5 m, and tamarisk forms dense thickets where the ground water lies from 1.5 to 6 m below the soil surface (Horton et al. 1960). Where ground water is deeper than 6 m, plants form an open shrubland (Horton and Campbell 1974).

Tamarisks have a wide tolerance of saline or alkaline soils (Robinson 1965), and Carmen and Brotherson (1980) found that tamarisk sites in Utah had higher soil salinity and pH than sites without tamarisk. Brotherson and Winkel (1986) identified the major factors that contribute to tamarisk success as alkaline soils, available soil moisture, and sufficient disturbance of native vegetation to facilitate tamarisk invasion. Ideal conditions for first-year survival for tamarisk seedlings are on gently sloping riverbanks, or sandbars and siltbars where water levels slowly recede during the period of seed fall (Everitt 1980).

Tamarisk is a highly fecund, relatively long-lived phreatophyte which is very tolerant of inundation, desiccation and nutrient stress (Stevens 1990). Tamarisk produces massive quantities of minute seeds that are readily dispersed by wind, and seeds are viable for up to 45 days under ideal conditions during summer. Once in contact with water, germination is completed within 24 hours following contract with water. Tamarisk seeds have no dormancy or after-ripening requirements.

Tamarisk flowers in two flushes, one in April-May and another in late July (in northern Arizona), presumably reflecting availability of spring high-water (e.g., linked to snowmelt) and summer moisture as rain. Tamarisk flowers continuously under favorable environmental conditions but the flowers required insect pollination to set seed. Tamarisk seed lived for only a few weeks during the summer; and the few seeds that might survive over winter under cooler conditions did not appear to form a persistent seed bank (Stevens 1990).

Tamarisk will produce roots from buried or submerged stems or stem fragments (Merkel and Hopkins 1957). Such a life history attribute allows tamarisk to produce new plants vegetatively following floods from stems torn from the parent plants and buried by sediment. Ideal conditions for first-year survival are saturated soil during the first few weeks of life, a high water table, and open sunny ground with little competition from other plants.

Hem (1967) studied the salts present in leaves and stems of *T. pentandra* at locations in Arizona and New Mexico. He found that the total concentration of calcium, magnesium, chloride, and sulfate in the leaves generally ranged from 5 to 15% of their dry weight. About 10% of the total ionic concentration consisted of inorganic ions that could be readily washed off the leaves by rainfall.

Native distribution. The family Tamaricaceae is native to Africa, Asia, and Europe (Robinson 1965), and the native range of the 5-merous tamarisk (*T. ramosissima*) is from the southern Europe to Asia minor and eastward to Mongolia, Tibet, central China and North Korea (Crins 1989). *T. parviflora*, the 4-merous tamarisk, has a native range lying in southern Europe, perhaps extending as far south as northern Algeria (Crins 1989). *T. aphylla* is a severe pest of riparian areas in arid central Australia where it impacts systems similarly to *T. ramosissima* and *T. parviflora* have in the southwestern U.S (Griffin et al. 1989); *T. aphylla* is not considered invasive in North America.

Non-indigenous distribution. Tamarisk has spread to all of the western and Great Plains states, with the greatest concentrations in Texas, Arizona and New Mexico (Robinson 1965; Figure 30). It is also abundant in California, Nevada, Utah and western Colorado. Wyoming and Montana have recently been invaded along with tributaries of the Missouri. North Dakota recorded its first observation of tamarisk in 2002 (Figure 31). It is not clear whether or not the 5-merous species (*T. ramosissima*) dominates in some areas and the 4-merous species (*T. parviflora*) in others. Both the 5-merous species and the 4-merous species also escape from cultivation occasionally in the eastern U.S., particularly on sandy beaches and roadsides, but are not invasive there (Gleason and Cronquist 1991, Radford et al. 1968, Wunderlin 1998). Weber (1990) reported that the Spanish explorer Father Escalante mentioned tamarisk in his journals from his travels throughout the American Southwest in 1776. If this is correct, it means that the Spanish introduced this species at least 200 years ago, although Robinson (1965) provided evidence that contradicts this claim. Robinson (1965) stated that tamarisk was offered for sale to the public in California beginning in the 1850s. Apparently, tamarisk did not start to become invasive in the U.S. until about 1877 when collections of tamarisk started to appear in herbaria (Robinson 1965). The plant did not attract much attention in the United States until the 1920s, and its impact on ground water was not appreciated until years later (Robinson 1965).

Impacts and control. Since the mid- to late 1800's, tamarisk has become naturalized along river bottoms and lake margins in the western United States, particularly in Arizona, New Mexico, California, Texas, Colorado, Utah, Nevada, Oklahoma and Wyoming. The species was first reported in North Dakota in 2002. There are multiple, interacting factors involved in the invasion of tamarisk (Everitt 1980), including intentional tamarisk plantings designed to protect streambanks and control erosion; conversion of native riparian forests to agricultural uses; damming of rivers fed by snowmelt which has shifted the time of peak discharge below the dams from spring to summer; creation of large areas of fine sediment that provide the ideal substrate for tamarisk colonization along the margins of reservoirs; increased salinity of rivers due to irrigation return flows and evaporation from reservoirs; reduced flood frequency downstream of reservoirs; and more stabilized base flows in rivers due to reservoir construction. Everitt (1980) noted that tamarisk has not become established in all western rivers, particularly those that still experience large floods and those where spring, rather than summer flooding still predominates. The spread of tamarisk has been and continues to be greatly facilitated by human activities.

Tamarisk possesses a number of highly desirable attributes for a successful invasive species, including an ability to 1) crowd out native stands of riparian and wetland vegetation; 2) increase the salinity of surface soil rendering the soil inhospitable to native plant species; 3) dries up springs, wetlands, riparian areas and small streams by lowering surface water tables; 4) widens

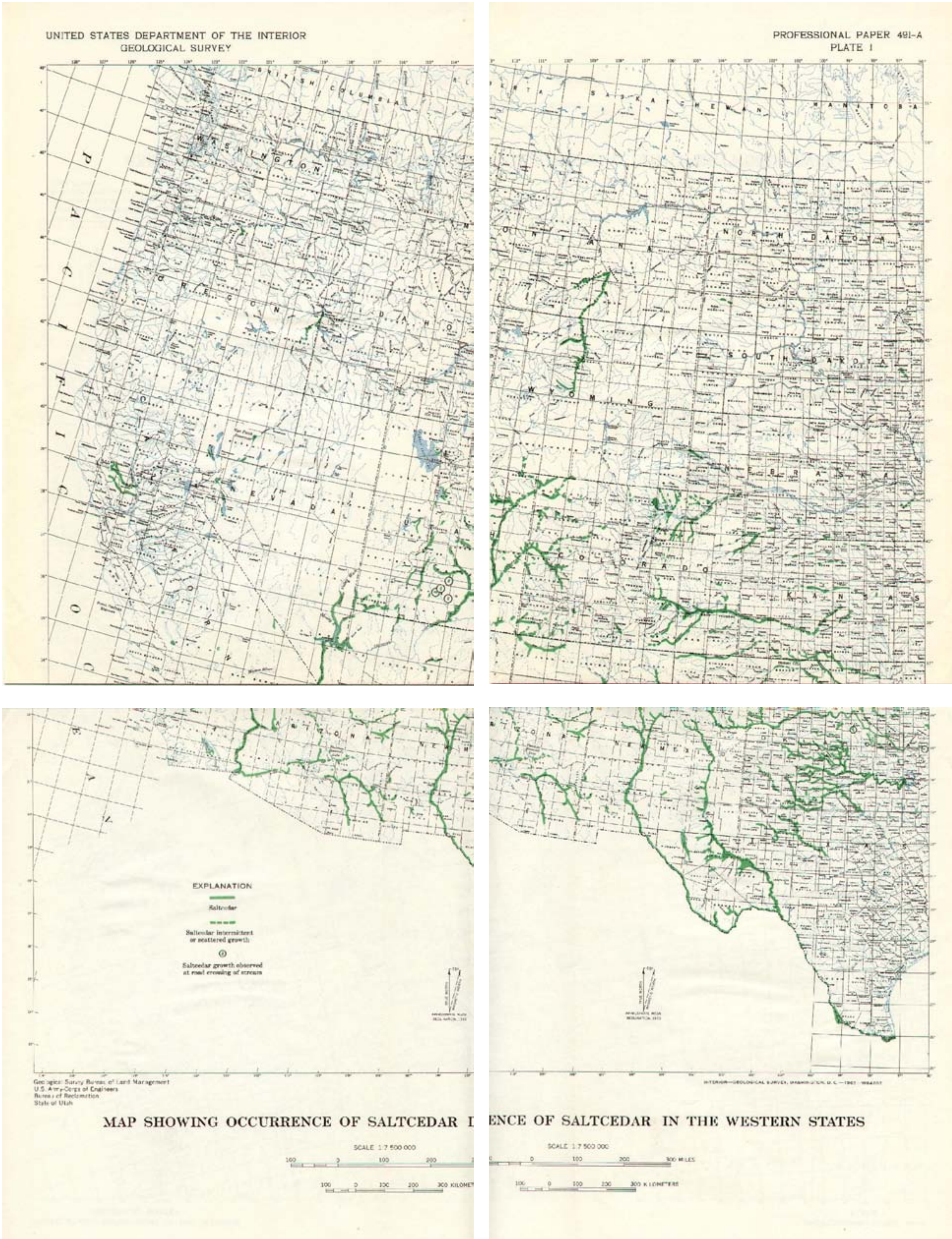
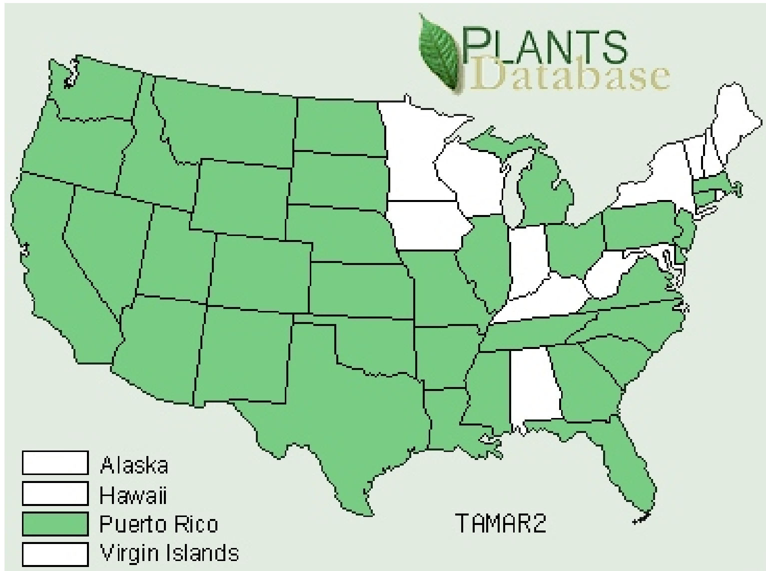


Figure 30. Sectional distribution map of *Tamarix* spp. as summarized in Robinson (1965).



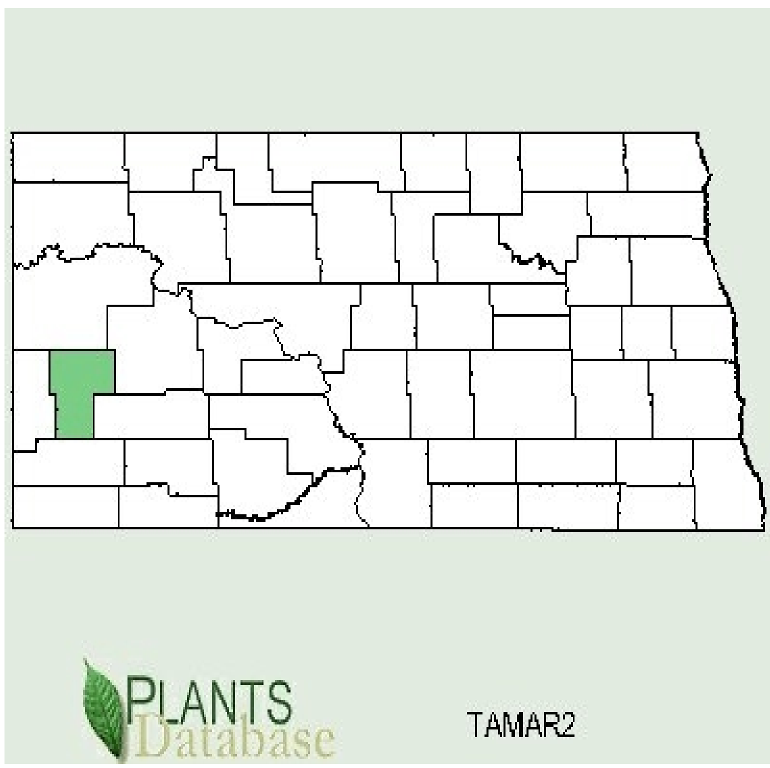


Figure 31. *Tamarix* spp. by state records (top) and county records (bottom). There are no confirmed observations of the plants in Canada.

floodplains by clogging stream channels; 5) increases sediment deposition due to the abundance of tamarisk stems in dense stands; and 6) uses more water than comparable native plant communities.

Crowding out native vegetation. A number of field observations clearly suggests that tamarisk can crowd out native riparian and wetland vegetation, yet human-induced changes in hydrologic regimes of rivers is likely not the only factor involved in the widespread invasion of tamarisk in the western US(Everitt 1980). Along some river courses in the southwest US, tamarisk is the dominant component in the vegetation community, e.g., along the lower Colorado River in Arizona and California, the elimination of flooding due to the construction of dams, the salinization of the soil and recurrent wildfires have virtually eliminated the cottonwood-willow riparian forests (R. D. Ohmart, personal communication). Tamarisk appears much less invasive along rivers where natural hydrologic processes are relatively intact. In many instances, tamarisk probably replaces rather than displaces native riparian vegetation, once native vegetation has been adversely effected by human activities. An outcome of a success invasion and the accompanying change in community structure is, wildlife habitat quality is generally diminished with increasing occurrence of tamarisk in the vegetation community.

Increasing salinity of surface soil. Tamarisk is a halophyte and actually mediates increases soil salinity. The plant's leaves and stems contain concentrations of soluble salts in the range of 5-15% (Hem 1967) which are absorbed by the roots from deeper soil layers, transported though the plant and concentrated in the leaves. With leaf fall, these salts are later deposited on the soil and become part of the soil litter.

Increased water consumption. Tamarisk stands in riparian ares translocate large amounts of ground water, an observation well documented nearly 40 years ago in a series of papers published by USGS (see Robinson 1965). Robinson (1965) cited studies which indicate tamarisk consumes on the order of 4 acre-feet of ground water annually. Sala et al. (1996) found that individual *Tamarix ramosissima* plants used about the same amount of water per unit of leaf area as did the native riparian species *Pluchea sericea*, *Prosopis pubescens* and *Salix exigua*, and their study confirmed previous work (Davenport et al.1982) that indicated evapotranspiration from riparian communities with high ground water availability is more dependent on stem density than on plant species composition. Sala et al. (1996) noted that tamarisk stands may have significantly more leaf area per unit of ground area than stands of native riparian vegetation. If so, the tamarisk stands would use more water per unit of ground area than the native stands and, replacing the tamarisk stands with native species would save water.

Weeks et al. (1987) presented data that suggested that conversion of stands of native riparian forest to a tamarisk stand may result in increased consumptive use of ground water. Few studies have demonstrated increases in ground water levels or stream flows in the southwestern US following tamarisk removal, except on a very local scale in small streams or springs. Many land managers, however, cite cases of springs that dried up following invasion by tamarisk, then tamarisk removal, springs flowed again (Barrows 1993). Brotherson et al. (1982) found that the proportion of xerophytic plant species increased as the age of tamarisk stands increased, which suggests that the longer a community had been occupied by tamarisk, the drier it became.

Widening floodplains and increasing deposition of sediment. Robinson (1965) claimed that dense stands of tamarisk could increase areas inundated by floods, because dense stands of tamarisk choked overflow and lateral channels, reducing the capacity of a stream channel and associated flood plain to transport flood waters. Dense stands of tamarisk also tended to increase sediment deposition due the increased channel roughness caused by tamarisk stems. Everitt (1980) noted that vegetation can promote local sediment deposition, but likely play little role in increasing larger-scale regional deposition of sediment.

Tamarisk is commonly controlled in riparian areas and wetlands and along lake shores because of its potential to displace native vegetation and its lower value as wildlife habitat. However, control over large areas is difficult in situations where hydrologic processes have been greatly altered, due to the high control cost and the likelihood that tamarisk will re-invade areas from which it is eliminated. Areas where tamarisk is to be managed should be selected carefully to maximize the likelihood of success.

Alternatives for control. Tamarisk has two traits that might be exploited for its control. First, tamarisk seedlings grow more slowly than many native riparian plant species. Second, mature tamarisk plants are highly susceptible to shading (Stevens 1990)

Management options for control of tamarisk reflect various considerations including the spatial extent of infestation (e.g., less than an acre to many acres), federal and state restrictions on chemical control agents, co-occuring native vegetation that should be maintained, the presence or absence of open water, adjacent land uses (e.g., potentially limiting chemical use or prescribed burns), and the availability and cost of labor.

Tamarisk can be controlled by three principal methods: chemical control, mechanical removal, and biologica control. Chemical control of tamarisk is the most frequently applied management tool, and often times chemical controls are used in conjunction with mechanical removals to gain long-term success in preventing spread and controlling existing stands of tamarisk.

Chemical controls and mechanical removal. Herbicide use may occur at various times during the plant's life, and depending on habitat, various management options are available. For example, chemcial controls may be implemented by: 1) applying herbicide to foliage of intact plants; 2) removing aboveground stems by burning or mechanical means followed by foliar application of herbicide; 3) cutting stems close to the ground followed by application of herbicide to the cut stems; 4) spraying basal bark with herbicide and 5) digging or pulling plants.

For larger areas (> 1 to 2 acres) that are essentially monotypic stands of tamarisk, the best methods would likely be foliar application of imazapyr (Arsenal®) herbicide to the intact plants or burning or cutting plants followed by foliar application of imazapyr or triclopyr (e.g. Garlon4® or PathfinderII®) to the resprouted stems. Foliar application of imazapyr or imazapyr in combination with glyphosate (e.g. Rodeo®) can be effective at killing large, established plants. Over 95% control has been achieved in field trials during the late summer or early fall. Herbicide application may be accomplished from the ground (hand-held or truck-mounted equipment) or from the air. Foliar application of herbicide works especially well in monotypic stands of tamarisk, although experienced persons using ground equipment can spray around native trees and shrubs such as cottonwood and willow. As an alternative to herbicides, prescribed fire or a bulldozer can be used to open up large stands of tamarisk. Once opened, the resprouts can be sprayed when they are 1 to 2 m tall using imazapyr, or imazapyr plus glyphosate, or triclopyr.

Tamarisk eradication in areas that contain significant numbers of interspersed, desirable shrubs and trees is difficult, and it may not be possible to rapidly kill tamarisk plants without also killing desirable shrubs and trees. Depending on the site, manual removal of plants may be necessary, then tamarisk stumps would be treated with herbicide. While labor costs are high with manual removal and subsequent herbicide treatments, desirable woody plants will be spared. Depending on the vegetation mix characterizing the habitat, an alternative approach to managing mixed-species eradication programs is to kill all woody plants at a site, then replant with desirable species.

For modest-sized areas (< 1 acre), the “cut-stump” method (i.e., cutting the stem and applying herbicide) is used in stands where woody native plants are present and where their continued existence is desired. Individual tamarisk plants are cut as close to the ground as possible with chainsaws, loppers or axes, and herbicide is applied immediately thereafter to the perimeters of the cut stems. Triclopyr (e.g. Garlon4® or PathfinderII®) and imazapyr (Arsenal®) can be very effective when used in this fashion, and treatments are most effective in the fall when plants are translocating materials to their roots. The efficacy of treatments is enhanced by cutting the stems within 5 cm of the soil surface, applying herbicide within one minute of cutting, applying herbicide all around the perimeter of the cut stems, and retreating any resprouts 4 to 12 months following initial treatment.

Long-term success at eradication, or at least control, of tamarisk depends on an effective initial treatment and periodic re-treatments, especially when 100% “kill” was not attained in a first round of treatment. To assure long-term management success, monitor and control programs must be designed and implemented to control tamarisk and reduce the likelihood that it will re-invade treated areas. If initial control efforts are effective, subsequent monitoring and surveillence programs are relatively inexpensive.

Control with burning. Tamarisk plants typically resprout vigorously after burning. However, burning followed by herbicide application to the resprouts can achieve excellent control in monotypic stands of tamarisk. Burning opens dense tamarisk stands and greatly reduces tamarisk biomass. Jorgensen (1996) recommended felling 20 to 25% of the largest tamarisk plants in stands several months prior to burning to create enough dry ground fuel to propagate a fire. Burning during the hottest part of the summer, when plants experience the greatest water stress, is likely to yield the best results. Chavez (1996), West (1996) and Egan (1996, 1997) used prescribed fire in Afton Canyon, California, to open dense stands of tamarisk for resprout treatment with herbicides. Duncan (1994) stated that repeated yearly burns can suppress tamarisk and kill some of the plants after 3 to 4 years.

Wildfire may be highly detrimental to native riparian forests, since it is a relatively novel disturbance in riparian forests of the southwestern US (Busch and Smith 1993). The dominant woody plant in many southwestern native riparian forests, Fremont cottonwood (*Populus fremontii* ssp. *fremontii*), does not re-sprout vigorously following fire, while tamarisk does. Busch (1995) concluded that the invasion of the alien tamarisk coupled with the novel disturbance of fire completely change southwestern riparian forests, based on his study of burned and unburned riparian forests along the lower Colorado River in Arizona. His results suggested that the native cottonwood – willow forest would be completely converted to tamarisk stands over the next several decades, and burning does not appear to be a reasonable control method for tamarisk where it occurs as a component of native communities.

Control with chemicals using foliar application to intact plants. Field studies in New Mexico by Duncan (1994) suggested that aerial application of the herbicide imazapyr (Arsenal®) alone or in combination with glyphosate (e.g. Roundup®, Rodeo®) is effective and practical for controlling tamarisk over thousands of hectares, particularly in dense stands where little on no native vegetation is present. Cost of aerial application of herbicide ranged from $70 to $90 per acre. Field trials along the Pecos River in New Mexico showed that fixed-wing aircraft could apply herbicide quite precisely, consistently following the 15 meter buffer line along the river bank. Several field trials have produced control rates of > 90% after one or two years. Alternatively, herbicide can be sprayed directly on tamarisk plants using truck-mounted equipment if stands are not too dense. This approach is appropriate where significant numbers of native trees and shrubs are interspersed with tamarisk plants. Duncan (personal communication) cautioned that sprayed plants should not be bulldozed or burned for two growing seasons, because disturbing the treated plants can induce some to resprout. Duncan and McDaniel (1996) have developed the following general guidelines:

- Focus treatment on young or regrowing tamarisk plants, because smaller plants are easier to kill than larger plants.
- Target areas previously plowed, mowed, burned or cleared, or areas where tamarisk appears to be invading.
- Target areas with tamarisk densities < 400 plants per hectare.

While the optimal herbicide proportions have not yet been developed, a mixture of 0.5% (v/v) imazapyr and 0.5% glyphosate (v/v) plus 0.25% (v/v) nonionic surfactant give satisfactory results. Kunzmann and Bennett (1990) stated that preliminary research indicates that the broad-spectrum herbicide imazapyr is the most cost-effective control technique known for tamarisk. However, they noted that more research is required to determine long-term effects of imazapyr on non-target plants and on other organisms.

Prescribed burning followed by foliar application of herbicide. This method is appropriate for larger areas, e.g., hundreds of hectares. Bureau of Land Management (BLM) has effectively used the method in the deserts of the southwestern US to control tamarisk and restore riparian vegetation (Egan 1996, 1997). Costs of removing the tamarisk and restoring native vegetation costs \$1500 to \$3000 per acre. As of 1997, tamarisk abundance had declined dramatically in the areas where it had been controlled (Egan 1997).

Cut-stump method. The cut-stump method is appropriate for modest-sized areas 2 hectares or smaller. Neill (1990, 1996) summarized the details of cut-stump herbicide treatments for tamarisk. Based on Neill's work, the triclopyr herbicides Garlon4® or PathfinderII® appear to be the best choices for killing tamarisk due to higher phytotoxicity, low toxicity to humans, lack of restriction, and cost comparable to the other herbicides when diluted as directed. These herbicides contain the same active ingredient, triclopyr. Garlon4® is diluted 1:3 (v/v) in the field with cheap vegetable oil while PathfinderII® is sold already mixed and diluted with vegetable oil. PathfinderII® also contains a dye, which makes it easier to distinguish stumps that have been treated from those that have not. Dyes such as colorfast® purple, colorfast® red and basoil® red can be added to Garlon4®.

Diluted, Garlon4® costs about \$26 per gallon, while PathfinderII® costs about \$30 per gallon. One gallon is sufficient to treat hundreds of cut stumps. Neill (1990, 1996) stated that 95% mortality can be expected with either of these herbicides, with lower mortality probably being the result of not cutting close enough to ground level and/or not treating the circumference of the stump completely. However, Howard (1983) found that cuts 15 to 30 cm above the ground surface were effective when using Garlon4® in the autumn. Neill (1990, 1996) noted that tamarisk plants are best located in the spring or summer when their pink flowers are visible, and that control during this period may be advisable even though the plants are less susceptible to the herbicide. Neither Garlon4® nor PathfinderII® is labeled for aquatic use; however, stumps located near but not in or over open water can be treated with these herbicides provided that none of the herbicide enters the water. Garlon3A®, an amine-based, water-soluble formulation of triclopyr, is registered for use "to control vegetation in and around standing water sites, such as marshes, wetlands, and the banks of ponds and lakes" (see Garlon 3A® label).

Control with chemicals using basal bark treatment with herbicide. Neill (1996) reviewed the basal bark method of tamarisk control. This method precludes the need to cut the tamarisk plants, resulting in major savings in labor and produces no tamarisk debris to haul away or burn. Disadvantages are the higher amount of herbicide required, up to five times that needed for stump-cut control, and lower mortality than with stump-cut. Neill (1996) noted that the basal bark method has been very effective at killing resprouts from debris piles left by a major flood. Jorgensen (1996) stated that basal bark application of Garlon4® was very effective on tamarisk plants with a basal diameter of less than 4 inches.

Carpet roller method. H.S. Mayeux with the USDA-ARS in Temple, Texas developed a carpet roller attachment for the front of a tractor. The roller is sprayed with herbicide, which is then applied to the tamarisk via the carpet roller as the tractor drives through the tamarisk stand. This method is an alternative in dense stands where desirable trees and shrubs are present. This method might also be useful in situations where standing water is interspersed with the tamarisk plants.

While a tabular summary of chemical control measures oversimplifies herbicide use for tamarisk control (see Table 5), these summary practices (see, e.g., Jackson (1996) for detail) provide starting points for use of various chemicals and their application to tamarisk control in western US.

Early reviews (Sisneros 1991) of herbicide control of tamarisk noted that Garlon® formulations were among the safest herbicides for mammals and other organisms, however, Garlon 4® is highly toxic to fish. Triclopyr, the active ingredient in all the Garlon® formulations decomposes rapidly after application, in less than one day in water and between 2 to 8 weeks in soil. Triclopyr will not kill grasses but it will kill native trees and shrubs.

Table 5. Summary of herbicide information relevant to tamarisk control (Jackson 1996).

Herbicide Trade Name	Active Ingredient	Formu-lation	Signal Word	Aquatic Registration	Foliar Applic?	Aerial Applic?	Stump Cut?	Basal bark Application
Arsenal®	Imazapyr	IPA-salt	Caution	No	Yes	Yes	Yes	No
Garlon 3A®	Triclopyr	Amine	Danger	No (applied for)	Yes	No	Yes	Yes
Garlon 4®	Triclopyr	Ester	Caution	No	Yes	No	Yes	Yes
Pathfinder II®	Triclopyr	Ester	Caution	No	No	No	Yes	Yes
Rodeo®	Glyphosate	IPA-salt	Caution	Yes	Yes	Yes	Yes	No
Roundup®	Glyphosate	IPA-salt	Caution	No	Yes	Yes	Yes	No

Control with cutting. A single cutting of tamarisk is ineffective, because tamarisks resprout vigorously. Cutting combined with herbicide treatment can be very effective at controlling tamarisk, and can reduce consumption of ground water through reduction of transpiring leaves. Resprouting from roots that remain is common, so repeated measures (e.g., cutting and burning, cutting and herbicide application) may be required to kill the root system.

Root plowing has been used to control tamarisk. It is important that the root plow cut the tamarisk root crowns well below the soil surface, e.g., 0.3 to 1.0 m. Root plowing works best during hot, dry conditions that help dry the cut roots. Root fragments left in the ground will often resprout after root plowing which means repeated measures to achieve control (e.g., hand-grubbing resprouts or spraying them with imazapyr or triclopyr). Root plowing is appropriate for large, dense stands that have little or no native vegetation and where prescribed burning or aerial application of herbicide is not feasible.

Control with grazing, dredging and draining. Tamarisk is able to extract water from deeper in the soil profile than the native species of cottonwood and willow. Therefore, draining and dredging that lead to local declines in water table depth could promote tamarisk at the expense of desirable native plants, rather than discourage tamarisk.

Cattle (and probably goats) will eat tamarisk, but grazing alone is probably not a feasible control method. However, goats might be able to control dense stands of tamarisk where little native vegetation is present, particularly if the stands are cut or burned first, with goats eating the regrowth.

Control with mowing, disking and pulling. Mowing might be a useful way to reduce the volume of tamarisk prior to treatment with herbicide, especially in relatively level sites where prescribed burning is not feasible. Hand pulling can be an effective way to control tamarisk in situations where the plants are small, where access is difficult, or where herbicides cannot be used. For example, hand pulling has been used to control new tamarisk plants around isolated desert springs in national parks after the larger tamarisk plants have been killed.

Biological control. In 1986, the U.S. Department of Agriculture's Agricultural Research Service (USDA-ARS) laboratory in Temple, Texas initiated a biological control program for tamarisk (DeLoach 1996). The goals for the program were to find and obtain insects that would damage *Tamarix ramosissima* without damaging native vegetation or *Tamarix aphylla*, the less invasive, evergreen species that is used for windbreaks and shade in the southwestern U.S. To date, two species of insect have been tested and proposed for release by USDA. One is a mealybug (*Trabutina mannipara*) from Israel and the other is a leaf beetle (*Diorhabda elongata*) from China. The leaf beetle defoliated tamarisks in greenhouse tests and the mealybug fed on twigs. DeLoach and Gould (1998) predict that these two insects may provide about 85% control of tamarisk and will take 3 to 5 years to control tamarisk at small sites and 5 to10 years to control tamarisks in small to medium watersheds.

Impacts. Early studies clearly suggested that tamarisk invasions of riparian habitats would be associated with adverse ecological effects. For example, Anderson et al. (1977) found that tamarisk stands along the lower Colorado River had lower bird density, bird species richness and diversity than did the native cottonwood-willow vegetation. Engel-Wilson and Ohmart (1978) found lower bird density and diversity in tamarisk stands along the lower Rio Grande River compared to native cottonwood-willow riparian forest. Kasprzyk and Bryant (1989) studied birds and small mammals along the Virgin River upstream from its inflow to Lake Mead in Nevada, and found that bird density and diversity were lower in tamarisk communities than native riparian vegetation. Ellis (1995) studied bird use of tamarisk and cottonwood vegetation in central New Mexico along the Rio Grande River where many bird species used both habitats, but three species used only tamarisk while six species using only cottonwood. Assuming the prediction by Howe and Knopf (1991) that tamarisk may completely supplant cottonwood habitat along the middle Rio Grande River in New Mexico over the next century, the richness of riparian bird species in that area would decline.

Brown and Johnson (1990) argued that, while tamarisk habitat along the lower Colorado River was much less valuable for breeding birds than native riparian habitat, the reverse was true along the Colorado River in Grand Canyon National Park. Hunter et al. (1988) proposed that bird nests in tamarisk along the lower Colorado River experienced higher heat loads than nests in multi-layered cottonwood forests that afford more shade. Anderson (1994) studied the Apache cicada in a native riparian community and a tamarisk stand along the lower Colorado River, and found that although cicadas were abundant in both communities, the insects emerged later in the native, cottonwood and willow-dominated communities when migrating and nesting birds were present. This change in temporal availability of this key food resource may help explain the low abundance of breeding birds in tamarisk communities.

Brown and Trosset (1988) stated that tamarisk stands in Grand Canyon National Park developed after construction of the of Glen Canyon Dam; comparable vegetation was not present along the river prior to construction of the Dam, so the tamarisk vegetation represented a new habitat type for that locale. In fact, black chinned hummingbirds (*Archilocus alexandri*) nested only in tamarisk-dominated habitats along the Colorado in the Grand Canyon (Brown 1992). Thus, Brown and Trosset (1988) argued that regional tamarisk management strategies must developed with respect to bird species.

Hunter et al. (1988) studied bird use in riparian vegetation along the middle Pecos River in New Mexico. There, birds used tamarisk as much as or more than other vegetation types year round. They noted that prior to invasion by tamarisk, this portion of the Pecos River had few tall, mature stands of vegetation. Thus, birds may have expanded their local ranges as tamarisk expanded. The lack of tall vegetation along the Pecos River contrasts with the condition of other desert riparian systems prior to Euro-American settlement (Ohmart and Anderson 1982).

The Federally Endangered Southwestern Willow Flycatcher (*Empidonax trailii extimus*) is known to nest in tamarisk-dominated areas (USFWS 1993). This subspecies of the Willow Flycatcher is widely distributed in scattered remnant populations across much of the area where tamarisk is invasive. Although it also feeds and breeds in riparian woodlands dominated by native plants including willows (*Salix* spp.) arrowweed (*Pluchea* spp.) and *Baccharis* species there has been concern that it might be further threatened if a biocontrol agent controls tamarisk over wide areas of the southwest. Others point out that even a highly successful biocontrol agent would not eliminate tamarisk and, that where it is reduced, native plants favored by breeding and feeding birds are likely to establish (Lovich and de Gouvenain 1998). Most published studies of the value of tamarisk to wildlife in North America have focused on birds and purported benefits to certain bird species may or may not extend to other animals (Lovich and de Gouvenain 1998).

Restoration potential. Smith and Devitt (1996) concluded that riparian restoration efforts that involve removing dense stands of tamarisk without restoring historical flow regimes will not be successful without extensive follow-up management. Native cottonwood and willow species may fail to re-establish without intensive planting in areas where floods have been eliminated or where receding flood flows do not occur when short-lived cottonwood and willow seeds are produced. Another potential problem is the ability of tamarisk to increase the salinity of surface soil due to deposition of highly saline leaf litter. In areas subject to frequent flooding, increased soil salinity should be a fairly transient phenomenon. High salinities may persist, however, in higher terraces along rivers whose banks are dominated by tamarisk because floodwaters rarely reach these areas. This may make it difficult or impossible for native plants to colonize these areas once tamarisk is controlled. Another problem may be downcutting of stream channels downstream of dams. In such situations, surface water tables may decline to the point that cottonwood and willows can no longer survive or colonize. Wildfire may be a problem because tamarisk-dominated communities experience higher fire frequencies than native cottonwood-willow communities, eventually eliminating the fire-sensitive cottonwood and perhaps even the willows (Busch 1995; Busch and Smith 1993). A final problem may be lack of a thorough network of mycorrhizal hyphae in soils that have been dominated by tamarisk for many years (St. John 1997). Mycorrhizae are important for many native species and their absence or low abundance may impede colonization of desirable plant species.

References

Anderson, B. W., A. Higgins, and R. D. Ohmart. 1977. Avian use of saltcedar communities in the Lower Colorado River Valley. USDA-Forest Service, General Technical Report RM-43:128-136.

Anderson, D. C. 1994. Are cicadas (*Diceroprocta apache*) both a “keystone” and a “critical link” species in lower Colorado River riparian communities? Southwest Naturalist 39(1):26-33.

Barrows, C. W. 1993. Tamarisk control II. A success story. Restoration and Management Notes 11(1): 35-38.

Baum, B. R. 1978. The genus *Tamarix*. Israel Academy of Science and Humanities, Jerusalem.

Bennett, P. S. and W. J. Burke 1990. Pp. 108-113 in: M. R. Kunzmann, R. R. Johnson and P. S. Bennett (eds.) Tamarisk control in southwestern United States. Proceedings of Tamarisk Conference, University of Arizona, Tucson, AZ, September 23-3, 1987. Special Report No. 9. National Park Service, Cooperative National Park Resources Studies Unit, School of Renewable Natural Resources, University of Arizona, Tucson, AZ.

Blaney and others. 1942. Consumptive use and requirements. Pp. 170-230 In: National Resources Planning Board, Pecos River Joint Investigation – Reports of the participating agencies. Washington, DC.

Bowman, C. 1990. 1987 tamarisk control project: Petrified Forest National Park. Pp. 11-16 in: M. R. Kunzmann, R. R. Johnson and P. S. Bennett (eds.) Tamarisk control in southwestern United States. Proceedings of Tamarisk Conference, University of Arizona, Tucson, AZ, September 23-3, 1987. Special Report No. 9. National Park Service, Cooperative National Park Resources Studies Unit, School of Renewable Natural Resources, University of Arizona, Tucson, AZ.

Brotherson, J. D., J. G. Carman and L. A. Szyska. 1982. Stem-diameter age relationships of *Tamarix ramosissima* in central Utah. Journal of Range Management 37:362-364.

Brotherson, J. D. and V. Winkel. 1986. Habitat relationships of saltcedar (*Tamarix ramosissima*) in central Utah. Great Basin Naturalist 46:535-541.

Brown, W. T. and R. R. Johnson. 1990. Ecology and management of riparian breeding birds in tamarisk habitats along the Colorado River in Grand Canyon National Park. Pp. 68-73 in: M. R. Kunzmann, R. R. Johnson and P. S. Bennett (eds.) Tamarisk control in southwestern United States. Proceedings of Tamarisk Conference, University of Arizona, Tucson, AZ, September 23-3, 1987. Special Report No. 9. National Park Service, Cooperative National Park Resources Studies Unit, School of Renewable Natural Resources, University of Arizona, Tucson, AZ.

Brown, B. T. and M. W. Trosset. 1989. Nesting-habitat relationships of riparian birds along the Colorado River in Grand Canyon, Arizona. Southwestern Naturalist 34:260-270.

Burke, W. J. 1990. Tamarisk and its control at Lake Mead National Recreation Area. P. 17-19 in: M. R. Kunzmann, R. R. Johnson and P. S. Bennett (eds.) Tamarisk control in southwestern

United States. Proceedings of Tamarisk Conference, University of Arizona, Tucson, AZ, September 23-3, 1987. Special Report No. 9. National Park Service, Cooperative National Park Resources Studies Unit, School of Renewable Natural Resources, University of Arizona, Tucson, AZ.

Busch, D. E. 1995. Effects of fire on southwestern riparian plant community structure. Southwestern Naturalist 40: 259-276.

Busch, D. E. and S. D. Smith. 1993. Effects of fire on water and salinity relationships of riparian woody taxa. Oecologia 94:186-194.

Carman, J. G. and J. D. Brotherson. 1982. Comparisons of sites infested and not infested with saltcedar (*Tamarix ramosissima*) and Russian olive (*Eleagnus angustifolia*). Weed Science 30:360-364.

Chavez, R. A. 1996. Integrated weed management: concept and practice. Pp. 32-36 in: J. Di Tomaso and C. E. Bell (eds.) Proceedings of the saltcedar management workshop, June 12, 1996, Rancho Mirage, CA. University of California Cooperative Extension Service, Holtville, CA.

Cinnamon, S. K. 1990. Wupatiki National Monument tamarisk and camelthorn eradication program 1983-88. Pp. 20-24 in: M. R. Kunzmann, R. R. Johnson and P. S. Bennett (eds.) Tamarisk control in southwestern United States. Proceedings of Tamarisk Conference, University of Arizona, Tucson, AZ, September 23-3, 1987. Special Report No. 9. National Park Service, Cooperative National Park Resources Studies Unit, School of Renewable Natural Resources, University of Arizona, Tucson, AZ.

Coffey, J. 1990. Summary report on tamarisk control: Joshua Tree National Park. Pp. 25-27 in: M. R. Kunzmann, R. R. Johnson and P. S. Bennett (eds.) Tamarisk control in southwestern United States. Proceedings of Tamarisk Conference, University of Arizona, Tucson, AZ, September 23-3, 1987. Special Report No. 9. National Park Service, Cooperative National Park Resources Studies Unit, School of Renewable Natural Resources, University of Arizona, Tucson, AZ.

Crins, W.J. 1989. The Tamaricaceae in the southeastern United States. Journal of the Arboretum 70:403-425.

Culler, R. C., R. L. Hansen, R. M. Myrick, R. M. Turner, and F. P. Kipple. 1982. Evaoptranspiration before and after clearing phreatophytes, Gila River floodplain, Graham County, Arizona. Geological Survey Professional Paper 655-P. U. S. Geological Survey, Reston, VA.

Davila, V. 1990. Tamarisk eradication efforts at Guadalupe Mountains National Park, Texas. Pp. 28-32 in: M. R. Kunzmann, R. R. Johnson and P. S. Bennett (eds.) Tamarisk control in southwestern United States. Proceedings of Tamarisk Conference, University of Arizona, Tucson, AZ, September 23-3, 1987. Special Report No. 9. National Park Service, Cooperative National Park Resources Studies Unit, School of Renewable Natural Resources, University of Arizona, Tucson, AZ.

DeLoach, C. J., and J. Gould. 1998. Biological control of exotic, invading saltcedars (*Tamarix* spp.) by the introduction of Tamarix-specific control insects from Eurasia. Proposal to USFWS, August 28 1998.

DeLoach, C. J., M. J. Pitcairn and D. Woods. 1996. Biological control of salt cedar in southern California. Pp. 30-31 in: J. Di Tomaso and C. E. Bell (eds.) Proceedings of the saltcedar management workshop, June 12, 1996, Rancho Mirage, CA. University of California Cooperative Extension Service, Holtville, CA.

Deuser, C. E. 1996. Salt cedar management: a success story. Pp. 50-52 in: J. DiTomaso and C. E. Bell (eds.) Proceedings of the saltcedar management workshop, June 12, 1996, Rancho Mirage, CA. University of California Cooperative Extension Service, Holtville, CA.

Duncan, K. W. 1994. Saltcedar: establishment, effects, and management. Wetland Journal 6(3):10-13.

Duncan, K. W. and K. C. McDaniel. 1996. Chemical weed and brush control guide for New Mexico rangelands. New Mexico State University Extension Service, Report 400 B-17, Las Cruces, New Mexico.

Egan, T. B. 1996. An approach to site restoration and maintenance for saltcedar control. Pp. 46-49 in: J. Di Tomaso and C. E. Bell (eds.) Proceedings of the saltcedar management workshop, June 12, 1996, Rancho Mirage, CA. University of California Cooperative Extension Service, Holtville, CA.

Egan, T. B. 1997. Afton Canyon riparian restoration project: fourth year status report. Presentation at tamarisk and Russian olive workshop, September, 1997, Grand Junction, CO.

Ellis, L. M. 1995. Bird use of saltcedar and cottonwood vegetation in the Middle Rio Grande Valley of New Mexico, U. S. A. Journal of Arid Environments 30:339-349.

Elzinga, C., D. Salzer, and J. Willoughby. 1998. Measuring and monitoring plant populations. Bureau of Land Management Technical Reference 1730-1.

Engel-Wilson, R. W. and R. D. Ohmart. 1978. Floral and attendant faunal changes on the lower Rio Grande between Fort Quitman and Presidio, Texas. Proceedings of the National Symposium for Protection and Management of Floodplain Wetlands. Pp. 139-147.

Everitt. B. L. 1980. Ecology of saltcedar – a plea for research. Environmental Geology 3:77-84.

Everitt, J. H., D. E Escobar, M. A, Alaniz, M. R. Davis and J. V. Richardson. 1996. Using spatial information techniques to map Chinese tamarisk (*Tamarix chinensis*) infestations. Weed Science 44: 194-201.

Fleming, C. M. 1990. Status report on Tamarix in Big Bend National Park. Pp. 33-35 in: M. R. Kunzmann, R. R. Johnson and P. S. Bennett (eds.) Tamarisk control in southwestern United States. Proceedings of Tamarisk Conference, University of Arizona, Tucson, AZ, September 23-3, 1987. Special Report No. 9. National Park Service, Cooperative National Park Resources Studies Unit, School of Renewable Natural Resources, University of Arizona, Tucson, AZ.

Gatewood, J. S., T. W. Robinson, B. R. Colby, J. D. Hem and L. C. Halfpenny. 1950. Use of water by bottom-land vegetation in lower Safford Valley, Arizona. Geological Survey Water Supply Paper 1103. U. S. Geological Survey, Reston, VA.

Gay, L. W. 1990. Saltcedar evapotranspiration and its measurement. Pp. 74-81 in: M. R. Kunzmann, R. R. Johnson and P. S. Bennett (eds.) Tamarisk control in southwestern United States. Proceedings of Tamarisk Conference, University of Arizona, Tucson, AZ, September 23-3, 1987. Special Report No. 9. National Park Service, Cooperative National Park Resources

Studies Unit, School of Renewable Natural Resources, University of Arizona, Tucson, AZ.

Gleason, H.A., and A. Cronquist. 1991. Manual of the vascular plants of the Northeastern United States and adjacent Canada, second edition. New York Botanical Garden. New York.
de Gouvenain, R. C. and B. R. West. E. 1996. Partnerships and volunteers for control of salt-cedar. Pp. 16-19 in: J. Di Tomaso and C. E. Bell (eds.) Proceedings of the saltcedar management workshop, June 12, 1996, Rancho Mirage, CA. University of California Cooperative Extension Service, Holtville, CA.

Griffin, G. F., D. M. Stafford Smith, S. R. Morton, G. E. Allen, and K. A. Masters. 1989. Status and implications of the invasion of tamarisk (Tamarix aphylla) on the Finke River, Northern Territory, Australia. Journal of Environmental Management 29:297-315.

Hays, F. and J. Mitchell. 1990 Pp. 36-38 in: M. R. Kunzmann, R. R. Johnson and P. S. Bennett (eds.). Tamarisk control in southwestern United States. Proceedings of Tamarisk Conference, University of Arizona, Tucson, AZ, September 23-3, 1987. Special Report No. 9. National Park Service, Cooperative National Park Resources Studies Unit, School of Renewable Natural Resources, University of Arizona, Tucson, AZ.

Hem, J. D. 1967. Composition of saline residues on leaves and stems of saltcedar (*Tamarisk pentandra* Pallas). Geological Survey Professional Paper 491-C. U. S. Geological Survey, Reston, VA.

Hoddenbach, G. 1990. Pp. 116-125 in: M. R. Kunzmann, R. R. Johnson and P. S. Bennett (eds.). Tamarisk control in southwestern United States. Proceedings of Tamarisk Conference, University of Arizona, Tucson, AZ, September 23-3, 1987. Special Report No. 9. National Park Service, Cooperative National Park Resources Studies Unit, School of Renewable Natural Resources, University of Arizona, Tucson, AZ.

Holland, J. S. 1990. Pp. 39-41 in: M. R. Kunzmann, R. R. Johnson and P. S. Bennett (eds.) Tamarisk control in southwestern United States. Proceedings of Tamarisk Conference, University of Arizona, Tucson, AZ, September 23-3, 1987. Special Report No. 9. National Park Service, Cooperative National Park Resources Studies Unit, School of Renewable Natural Resources, University of Arizona, Tucson, AZ.

Horton, J. S., F. C. Mounts, and J. M. Kraft. 1960. Seed germination and seedling establishment of phreatophytic species. Research Paper RM-48. USDA-Forest Service, Rocky Mountain Forest and Range Experiment Station, Ft. Collins, CO.

Horton, J. S. and C. J. Campbell. 1974. Management of phreatophytic and riparian vegetation for maximum multiple use values. Research Paper RM-117, USDA-Forest Service, Rocky Mountain Forest and Range Experiment Station, Ft. Collins, CO.

Howard, S. W., A. E. Dirar, J. O. Evan, and F. D. Provenza. 1983. The use of herbicides and/or fire to control saltcedar (*Tamarix*). Proceedings of the Western Society of Weed Science 36:65-72.

Howe, W. H. and F. L. Knopf. 1991. On the imminent decline of Rio Grande cottonwoods in central New Mexico. Southwestern Naturalist 36: 218-224.

Hunter, W. C., R. C. Ohmart and B. W. Anderson. 1988. Use of exotic saltcedar (*Tamarix chinensis*) by birds in arid riparian systems. Condor 90:11-1123.

Jackson, L. E. 1996. Chemical control of saltcedar (*Tamarix ramosissima*). Pp. 21-27 in: J. DiTomaso and C. E. Bell (eds.) Proceedings of the saltcedar management workshop, June 12, 1996, Rancho Mirage, CA. University of California Cooperative Extension Service, Holtville, CA.

Johnson, G. E. 1985. Salt cedar study in the Petrified Forest National Park. In: E. H. Colbert and R. R. Johnson (eds.) the Petrified Forest through the ages. Museum of Northern Arizona Bulletin Series #54.

Johnson, S. 1986. Alien plants drain western waters. Nature Conservancy News. September-October, 1986.

Jorgensen, M. C. 1996. The use of prescribed fire and mechanical removal as means of control of tamarisk trees. Pp. 28-29 in: J. DiTomaso and C. E. Bell (eds.) Proceedings of the saltcedar management workshop, June 12, 1996, Rancho Mirage, CA. University of California Cooperative Extension Service, Holtville, CA.

Kasprzyk, M. J. and G. L. Bryant. 1989. Results of biological investigations from the lower Virgin River vegetation management study. Bureau of Reclamation, Boulder City, NV. 75 pp.

Kerpez, T. A. and N. S. Smith. 1987. Saltcedar control for wildlife habitat improvement in the southwestern United States. U. S. Fish & Wildlife Service, Resource Publication 169, Washington, DC. 16 pp.

Kunzmann, M. R., R. R. Johnson and P. S. Bennett (eds.) 1990. Tamarisk control in southwestern United States. Proceedings of Tamarisk Conference, University of Arizona, Tucson, AZ, September 23-3, 1987. Special Report No. 9. National Park Service, Cooperative National Park Resources Studies Unit, School of Renewable Natural Resources, University of Arizona, Tucson, AZ.

Kunzmann, M. R. and P. S. Bennett. 1990. Arsenal as a control agent for saltcedar (Tamarisk). Pp. 82-90 in: M. R. Kunzmann, R. R. Johnson and P. S. Bennett (eds.) Tamarisk control in southwestern United States. Proceedings of Tamarisk Conference, University of Arizona, Tucson, AZ, September 23-3, 1987. Special Report No. 9. National Park Service, Cooperative National Park Resources Studies Unit, School of Renewable Natural Resources, University of Arizona, Tucson, AZ.

Lovich, J. E. and R. C. de Gouvain. 1998. Saltcedar invasion in desert wetlands of the southwestern United Sates: ecological and political implications. In: S. K. Majumdar et al. (eds.) Ecology of wetlands and associated systems. Pennsylvania Academy of Science, Philadelphia, PA.

Luttrell, C. 1983. Tamarisk control study. Unpublished report. Lake Mead National Recreation Area, Boulder, City, NV.

Merkel, D. L. and H. H. Hopkins. 1957. Life history of saltcedar. Transactions of the Kansas Academy of Science 60:360-369.

Mikus, B. 1990. Pp. 42-45 in: M. R. Kunzmann, R. R. Johnson and P. S. Bennett (eds.) 1990. Tamarisk control in southwestern United States. Proceedings of Tamarisk Conference, University of Arizona, Tucson, AZ, September 23-3, 1987. Special Report No. 9. National Park Service, Cooperative National Park Resources Studies Unit, School of Renewable Natural Resources, University of Arizona, Tucson, AZ.

Neill, W. M. 1990. Pp. 91-98, In: M. R. Kunzmann, R. R. Johnson and P. S. Bennett (eds.) Tamarisk control in southwestern United States. Proceedings of Tamarisk Conference, University of Arizona, Tucson, AZ, September 23-3, 1987. Special Report No. 9. National Park Service, Cooperative National Park Resources Studies Unit, School of Renewable Natural Resources, University of Arizona, Tucson, AZ.

Neill, W. M. 1996. Putting it altogether: management strategies and implementation. Presentation at Saltcedar Management and Riparian Restoration Workshop, Las Vegas, NV, September, 1996.

Ohmart, R. D. and B. W. Anderson. 1982. North American desert riparian ecosystems. Pp. 433-479 in: G. L. Bender (ed.) Reference handbook on the deserts of North America. Greenwood Press, Westport, CT.

Radford, A.E., H.E. Ahles and C.R. Bell. 1968. Manual of the vascular flora of the Carolinas. University of North Carolina Press, Chapel Hill.

Robinson, T. W. 1965. Introduction, spread and areal extent of saltcedar (*Tamarix*) in the Western States. Geological Survey Professional Paper 491-A. U. S. Geological Survey, Reston, VA.

Rowlands, P. G. 1990. History and treatment of the saltcedar problem in Death Valley National Monument. Pp. 46-56 in: M. R. Kunzmann, R. R. Johnson and P. S. Bennett (eds.) Tamarisk control in southwestern United States. Proceedings of Tamarisk Conference, University of Arizona, Tucson, AZ, September 23-3, 1987. Special Report No. 9. National Park Service, Cooperative National Park Resources Studies Unit, School of Renewable Natural Resources, University of Arizona, Tucson, AZ.

Sala, A., S. D. Smith and D. A. Devitt. 1996. Water use by *Tamarix ramosissima* and associated phreatophytes in a Mojave Desert floodplain. Ecological Applications 6:888-898.

Sharrow, D. 1990. Status of tamarisk and its control in Grand Canyon National Park. Pp. 57-60 in: M. R. Kunzmann, R. R. Johnson and P. S. Bennett (eds.) Tamarisk control in southwestern United States. Proceedings of Tamarisk Conference, University of Arizona, Tucson, AZ, September 23-3, 1987. Special Report No. 9. National Park Service, Cooperative National Park Resources Studies Unit, School of Renewable Natural Resources, University of Arizona, Tucson, AZ.

Sisneros, D. 1991. Herbicide analysis: Lower Colorado River saltcedar vegetation management study. Bureau of Reclamation, Denver, CO 165 pp.

Smith S. D. and D. A. Devitt. 1996. Physiological ecology of saltcedar: why is it a successful invader? Presentation at Saltcedar Management and Riparian Restoration Workshop, Las Vegas, NV, September, 1996.

Stein, E. D. 1996. Chemical control of saltcedar (*Tamarix ramosissima*). Pp. 21-27 in: J. Di Tomaso and C. E. Bell (eds.) Proceedings of the saltcedar management workshop, June 12, 1996, Rancho Mirage, CA. University of California Cooperative Extension Service, Holtville, CA.

Stevens, L. E. 1990. Pp. 99-105 in: M. R. Kunzmann, R. R. Johnson and P. S. Bennett (eds.) Tamarisk control in southwestern United States. Proceedings of Tamarisk Conference, University of Arizona, Tucson, AZ, September 23-3, 1987. Special Report No. 9. National Park Service, Cooperative National Park Resources Studies Unit, School of Renewable Natural Resources, University of Arizona, Tucson, AZ.

St. John, T. 1996. Establishing functional riparian communities after saltcedar removal. Presentation at Saltcedar Management and Riparian Restoration Workshop, Las Vegas, NV, September, 1996.

Sudbrock, A. 1993. Tamarisk control. I. Fighting back – An overview of the invasion, and a low-impact way of fighting it. Restoration and Management Notes 11(1): 31-34.

Taylor, J. P. 1996. Saltcedar management and riparian restoration. Presentation at Saltcedar Management and Riparian Restoration Workshop, Las Vegas, NV, September, 1996.

Thomas, L., K. Kitchell and T. Graham. 1990. Summary of tamarisk control efforts at Canyonlands and Arches National Parks and Natural Bridges National Monument. Pp. 61-66 in: M. R. Kunzmann, R. R. Johnson and P. S. Bennett (eds.) Tamarisk control in southwestern United States. Proceedings of Tamarisk Conference, University of Arizona, Tucson, AZ, September 23-3, 1987. Special Report No. 9. National Park Service, Cooperative National Park Resources Studies Unit, School of Renewable Natural resources, University of Arizona, Tucson, AZ.

Turner, R. M. 1974. Quantitative and historical evidence of vegetation changes along the Upper Gila River, Arizona. Professional Paper 655-H. U. S. Geological Survey, Reston, VA.
U. S. Bureau of Reclamation. 1973. Progress report, phreatophyte investigations, Bernardo evapotranspirometers. U. S. Bureau of Reclamation, Middle Rio Grande Project Office.

USFWS (U.S. Fish and Wildlife Service). 1993. Endnagered and threatened wildlife and plants. proposed rule to list the Southwestern Willow Flycatcher as endangered with critical habitat. Federal Register 58:39495-39522.

Van Hylckama, T. E. A. 1974. Water use by saltcedar as measured by the water budget method. Geological Survey Professional Paper 491–E. U. S. Geological Survey, Reston, VA.
Weber, W. A. 1990. Colorado Flora – Eastern Slope. University of Colorado Press, Niwot, Colorado.

Weeks, E. P., H. L. Weaver, G. S. Campbell and B. N. Tanner. 1987. Water use by saltcedar and by replacement vegetation in the Pecos River floodplain between Acme and Artesia, New Mexico. Geological Survey Professional Paper 491-G. U. S. Geological Survey, Reston, VA.

Welsh, S. A., N. D. Atwood, L. C. Higgins and S. Goodrich. 1987. A Utah Flora. Great Basin Naturalist Memoir Number 9. Brigham Young University, Orem, UT. 894 pp.

West, B. R. 1996. Prescribed burning and wildlife (fire as a management tool in saltcedar management). Presentation at Saltcedar Management and Riparian Restoration Workshop, Las Vegas, NV, September, 1996.

Wilgus, F. and K. L. Hamilton. 1962. Germination of saltcedar seed. Weeds 10(4):332-333.

Willits, P. 1994. Unpublished report on tamarisk treatments on the San Miguel preserve. The Nature Conservancy, Colorado Field Office.

Wunderlin, R.P. 1998. Guide to the vascular plants of Florida. University Press of Florida, Gainesville.