CLUBSHELL *(Pleurobema clava)* and NORTHERN RIFFLESHELL *(Epioblasma torulosa rangiana)*

RECOVERY PLAN

Prepared by:

G. Thomas Watters
Division of Wildlife
Ohio Department of Natural Resources
Columbus, Ohio

for

Region Five
U.S. Fish and Wildlife Service
Hadley, Massachusetts

Approved: Ronald E. Lambertson
Regional Director, U.S. Fish and Wildlife Service Region Five

Date: Sept 21, 1994

EXECUTIVE SUMMARY

Clubshell and Northern Riffleshell Draft Recovery Plan

CURRENT STATUS: The clubshell (*Pleurobema clava*) and northern riffleshell (*Epioblasma torulosa rangiana*) were once widespread throughout most of the Ohio River and Maumee river drainages, and the clubshell appears to have been very common. Both species now exist in eight to ten isolated populations each, most of which are small and peripheral. The largest remaining population of the clubshell is in the Tippecanoe River of Indiana; that of the northern riffleshell is in French Creek, Pennsylvania. The clubshell and northern riffleshell are threatened by runoff and channelization, domestic and commercial pollution, in-stream sand and gravel mining, impoundment, and zebra/quagga mussel infestation. Both species were Federally listed as endangered in February 1993.

HABITAT REQUIREMENTS AND LIMITING FACTORS: The clubshell is found in clean, coarse sand and gravel in runs, often just downstream of a riffle. It cannot tolerate mud or slackwater conditions, and is very susceptible to siltation. The riffleshell also occurs in packed sand and gravel in riffles and runs. More specific biology of these species largely is unknown, although general accounts of unionid biology may be applicable.

RECOVERY OBJECTIVES: The primary objective of the recovery program is to maintain and restore viable populations of both species to a significant portion of their historical range, thereby enabling reclassification and eventual delisting.

RECOVERY CRITERIA: To reclassify, viable populations must be established in 10 drainages for each species; these populations should include both peripheral and central populations to maintain whatever fraction of original genetic variability is left. To delist, each of the above 20 populations must be extensive and abundant enough to survive a single adverse ecological event, and the populations and their drainages must be permanently protected from all foreseeable threats.

ACTIONS NEEDED:

1. Initiate and participate in ecosystem conservation efforts.
2. Protect and manage mussel populations and their habitat on a site-specific basis.
3. Collect data on both species that are necessary for their recovery.
4. As needed, restore habitats and reintroduce the species to suitable areas.
5. Enlist public support for the recovery process through an outreach program and incentives.

ESTIMATED COSTS (in thousands):

	NEED 1	NEED 2	NEED 3	NEED 4	NEED 5	TOTAL
FY1	87	110	30		10	237
FY2	72	110	112		17	311
FY3	66	110	112	60	9	357
FY4-25	300	675	60	670	112	1817
TOTAL	525	1005	314	730	148	2722

DATE OF RECOVERY: Contingent on implementing recovery tasks on schedule, full recovery is anticipated by the year 2020.

* * * * *

The following recovery plan delineates actions required to recover and/or protect the endangered clubshell *(Pleurobema clava)* and northern riffleshell *(Epioblasma torulosa rangiana)*. Attainment of recovery objectives and availability of funds will be subject to budgetary and other constraints affecting the parties involved, as well as the need to address other priorities.

This approved plan has been prepared through contract with G. Thomas Watters of the Ohio Department of Natural Resources, with input from other resource experts and cooperators. The document does not, however, necessarily represent the views or official position of any individuals or agencies involved in its formulation, other than the U.S. Fish and Wildlife Service. Approved recovery plans are subject to modification as dictated by new findings, changes in species status, and the completion of recovery tasks.

Literature citations should read as follows:

U.S. Fish and Wildlife Service. 1994. Clubshell *(Pleurobema clava)* and Northern Riffleshell *(Epioblasma torulosa rangiana)* Recovery Plan. Hadley, Massachusetts. 68 pp.

Additional copies of this plan can be purchased from:

Fish and Wildlife Reference Service
5430 Grosvenor Lane, Suite 110
Bethesda, Maryland 20814
301/492-6403 or 1-800-582-3421

Cost varies according to number of pages.

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PART I: INTRODUCTION

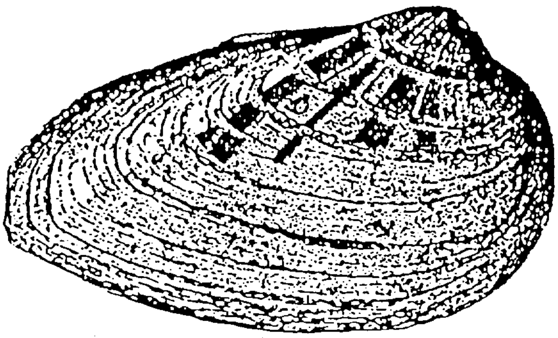
The clubshell (*Pleurobema clava*) and northern riffleshell (*Epioblasma torulosa rangiana*) were listed as endangered species on 22 February 1993 (50 CFR 17). Both were widespread throughout most of the Ohio River and Maumee River drainages prior to 1800. The clubshell was apparently very common. These species now exist in eight to ten isolated populations each, most of which are small and peripheral. The largest remaining clubshell population is in the Tippecanoe River of Indiana, and that of the northern riffleshell is in French Creek, Pennsylvania. Both species are threatened by runoff and channelization, domestic and commercial pollution, in-stream sand and gravel mining, impoundment, and zebra/quagga mussel infestation.

DESCRIPTION AND TAXONOMY

PLEUROBEMA CLAVA

The clubshell was described by Lamarck in 1819 as *Unio clava*, from a specimen sent to him labeled "Lake Erie." Simpson (1900: 737) believed this species was "one of the most striking of North American Uniones." The clubshell usually is easy to identify, even from fragments, within most of its range (Figure 1).

Based on growth annuli, the clubshell may live for 20 years or more. Shells grow to about 3 inches in length, averaging 1 to 1.5 inch. It is relatively light when juvenile, becoming more massive with age. Umbos are prominent, placed far anterior, sometimes projecting beyond the anterior margin of the shell lip, and become more anteriorly placed with age. There is no sexual dimorphism in the shell, which is distinctly trigonal or wedge shaped (the specific name *clava* is from the Latin for "club"). Most specimens are distinctly longer than high, although there is great variation in this feature. A sulcus may be present in old individuals. Beak sculpture consists of a series of small, weak nodes on the dorsal ridge, usually eroded away except in the youngest specimens. The shell is colored straw-yellow or light brown, with distinct green rays. The rays may be thick blotches or thin lines, usually interrupted at growth lines, and may become obsolete on old individuals.



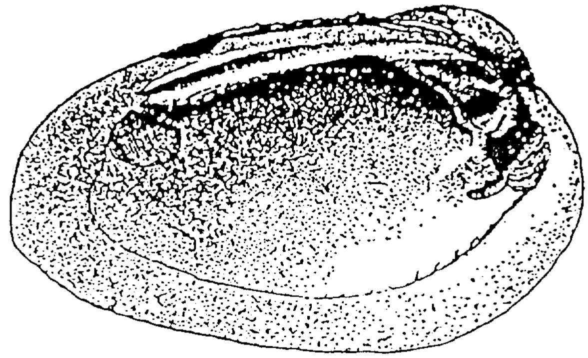
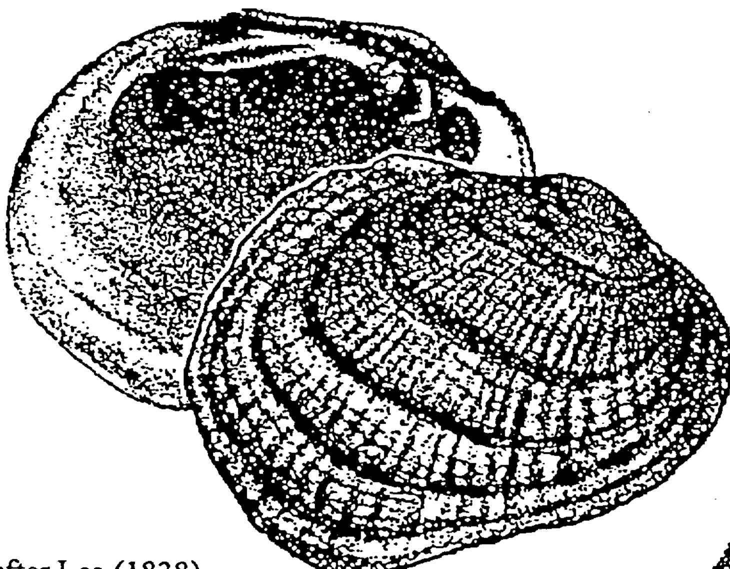
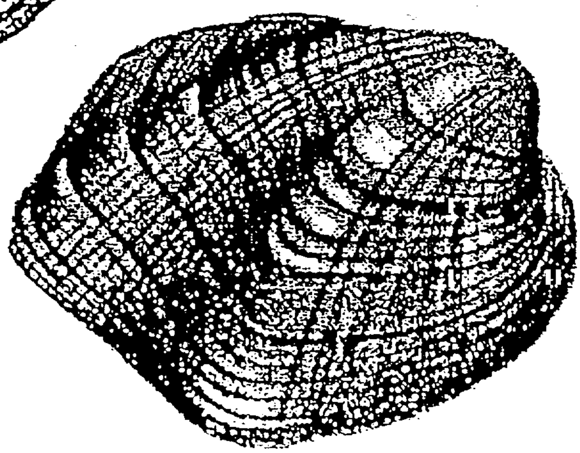


Figure 1.
Pleurobema clava (Lamarck, 1819)
After Call (1882)



Female, after Lea (1838)

Figure 2.
Epioblasma torulosa rangiana (Lea, 1837)



Male, after Conrad (1836)

The hinge and teeth are well developed, with a long, slightly arched lateral and short, chunky cardinals. The beak cavity is shallow and open. The nacre is white, tending to be iridescent posteriorly, particularly in juveniles.

Lea (1863) and Ortmann (1912) described the soft parts; Ortmann's comments are repeated here (pp. 264, 265):

> "Anatomy like that of the other species of *Pleurobema*. It should be mentioned that the mantle-connection between the anal and supra-anal is rather short, and was always found present. The anal is rather distinctly, but finely, papillose. Posterior margins of palpi connected only for a short distance.
>
> The outer gills alone are marsupial, and the placentæ are rather distinct. Glochidia of small size, subovate, without hooks. Their length and height is about the same, 0.16 mm.
>
> Color of soft parts whitish, with foot and gills grayish, and the margin of the mantle black posteriorly. In other specimens the foot is pale orange, as are also the margins of the mantle and adductors. The gills are grayish brown. There are all intergrades between these extremes. The placentæ are white, cream-color, or pale orange."

This species is difficult to separate from the Cumberlandian analog *Pleurobema oviforme*, which may represent a set of sibling taxa. Specimens from the Cumberland and lower Tennessee Rivers are virtually indistinguishable from some northern *clava*, and may represent that species. However, *P.* cf. *oviforme* is extirpated from much of that region, although it still exists in the Duck River and tributaries (Little South Fork, Buck Creek, Rockcastle River system, and others) of the upper Cumberland River (R. Anderson, Indiana Department of Natural Resources, *in litt.* 15 December 1993).

EPIOBLASMA TORULOSA RANGIANA

Lea (1837) described this species, named after the French malacologist Sander Rang, based on a female shell (Figure 2). He gave the type locality as "Ohio River, near Cincinnati" and "near Poland, Ohio." The latter is on Yellow Creek of the Mahoning River. There has been considerable confusion as to the name of this and closely related taxa. Many authors have considered it a headwater form or subspecies of *Epioblasma torulosa torulosa*, and it is so listed by the U.S. Fish and Wildlife Service. Others consider it a distinct headwater species

(D. Stansbery, Ohio State University Museum of Zoology, pers. comm. 10 August 1993). Older records may refer to this taxon as *delicata* or *perplexa*, but these names frequently were used for other species as well. Johnson (1978: 261) erroneously synonymized perobliqua with this group, having based his decision on the wrong illustration. His comments refer to Conrad's (1836) figure 1 (*torulosa*), not figure 2 (*perobliqua*). This taxon also may be listed under the genera *Dysnomia*, *Truncilla*, or *Plagiola*.

Based on growth annuli, the northern riffleshell may live for 15 years or more. Shells grow to about 3 inches in length, averaging 1.5 inches, and are relatively light but sturdy. Umbos are placed approximately 1/4 to 1/3 back from the anterior margin. The shells are distinctly sexually dimorphic.

The males are twice as long as high with a distinct sulcus. The area just preceding the sulcus is raised, often with very weak undulations corresponding to growth annuli. The dorsal slope ends in a blunted point at the posterior margin at about half the height of the shell. Umbos are low but distinct. Beak sculpture consists of a series of double loops, usually eroded away except in the youngest specimens. The beak cavity is shallow and open. The shell is colored straw-yellow, light brown, or greenish, with distinct dark green rays. The rays are thin lines, not interrupted at growth lines, and persist onto old individuals.

The male hinge and teeth are distinct, with a long, nearly straight lateral, and small chunky cardinals. The nacre is white, iridescent posteriorly.

The female shell is greatly expanded posteriorly; however, the degree of inflation is variable. This expansion begins to appear on the shell by the third year. Younger female shells resemble male shells. The growth annuli often form coarse concentric undulations over the expansion. The periostracum may project past the shell margins in the expanded area and is frequently thin and easily broken. Umbos and shell color are as in the males.

The female hinge usually has distinct teeth, but these become obliterated in some specimens. Lateral teeth are short and slightly arched, the cardinals short and chunky. The nacre is white, iridescent posteriorly. Rare specimens (both male and female) may have a rose or orange background color, as well as a rose-flushed nacre. These colors often fade after death.

Lea (1863) and Ortmann (1912) have described the soft parts. The latter account is given below (pp. 358, 359):

"The marsupium is greatly swollen, rather low and long, not so much deformed. Glochidia are also similar; length 0.26; height 0.23 mm., but my measurements are not very accurate, since all the glochidia I have are very young and delicate.

In the female, the two edges of the mantle diverge greatly in front of the branchial, the outer one curving outward, and forming a great, almost semicircular lobe, with a smooth edge; while the inner one runs almost straight downward and forward; the two edges coming together again at about the middle of the lower margin. The inner edge has crowded, very fine papillæ, which decrease anteriorly, and the anterior part of the edge is smooth. The space between the two edges is of a peculiar spongy structure, full of what appear as finely rounded or elongated pores.

In the male the two edges of the mantle are subparallel and close together, as usual, and the inner one has very minute papillæ.

The color of the soft parts is generally whitish or yellowish white. Outer edge of mantle grayish posteriorly, in the region of the anal and supra-anal blackish, not spotted. Papillæ of branchial brown, but this color does not run forward along the inner edge, and the inner edge itself and the spongy space between the two edges is snow-white."

Species that may be confused with *E. t. rangiana* include *E. t. torulosa*, *E. t. gubernaculum*, and *E. obliquata perobliqua*. Some specimens of *rangiana* are difficult to separate from the larger river form of subspecies *torulosa*. Generally, in *t. torulosa* the area immediately anterior to the sulcus is distinctly knobbed, often having a contorted appearance, in both sexes. It is much smoother and less prominent in *rangiana*. The marsupial expansion of the shell in *torulosa* is more posteriorly expanded and quite laterally compressed. This region in *rangiana* is not as posteriorly expanded and is often more laterally inflated. *Torulosa* often is colored a dark bluish-green, not light brown, green, or yellow as in *rangiana*. Males of *Epioblasma obliquata perobliqua* are very similar to *rangiana* males, although *Perobliqua* is not as angular, and the umbos are placed more anteriorly. Females of the two species are not at all similar. The Cumberlandian headwater form or subspecies of *torulosa*, *gubernaculum*, also is similar to *rangiana*. The marsupial region of that taxon is more expanded, like *torulosa*, but lacks the contorted knobs. It also is colored dark green.

DISTRIBUTION

Pleurobema clava and *Epioblasma torulosa rangiana* primarily are upper Ohio River system species. Records from the lower Tennessee and Cumberland River systems are probably authentic, although the clubshell has been confused with *P. oviforme* from those rivers (D. Stansbery, OSUMZ, pers. comm. 10 August 1993; R. Anderson, INDNR, *in litt.* 15 December 1993). The historic and present ranges of both species are shown in Figures 3-6. Historical data are those for which no living or freshdead individuals have been seen in the past decade. Data for these figures were derived from collection records of the University of Michigan Museum of Zoology, the Ohio State University Museum of Zoology, the Illinois Natural History Survey, West Virginia Department of Natural Resources, Dr. Arthur Bogan, the Western Pennsylvania Conservancy, the Michigan Natural Features Inventory, the Ohio Heritage Program, the Kentucky Nature Preserves Commission, Ecological Specialists, Inc., and published records. These maps show only records with relatively specific localities, and some points may be approximate. A more general listing, including vague records, is given in Table 1.

PLEUROBEMA CLAVA

The clubshell is an Ohio River system species recorded from most of the tributaries in Kentucky, Illinois, Indiana, and Ohio, as well as from more isolated systems in Michigan, Pennsylvania, and West Virginia. Records of *Pleurobema clava* from Nebraska (Aughey 1877) and Minnesota and Iowa (Simpson 1900) are erroneous (*fide* Grier and Mueller 1922), as is an Ottawa River record (*fide* La Rocque 1953). Some records from the Cumberland River appear to be authentic, but others may represent *Pleurobema oviforme* (see Gordon and Layser 1989, Cicerello *et al.* 1991). The type locality of Lake Erie generally has been regarded as spurious, but the University of Michigan Museum of Zoology has a specimen from La Plaisance Bay, Michigan, collected by Goodrich (no date, UMMZ 107494).

Subfossil shells clearly show that this species was once widespread and abundant (Dean 1890, Watters 1988a). Lemon (1898: 7) stated that it was "extremely common." Although now considered a creek or small river species, many records from larger rivers such as the Wabash and Tennessee show that this is a recent misconception.

The largest extant population is in the Tippecanoe River, Indiana (Cummings and Berlocher 1990; Cummings *et al.* 1992; ESI 1992, 1993a). Surveys by ESI in 1992 and 1993 found living individuals at nine sites from the mouth to the uppermost reach, a distance of

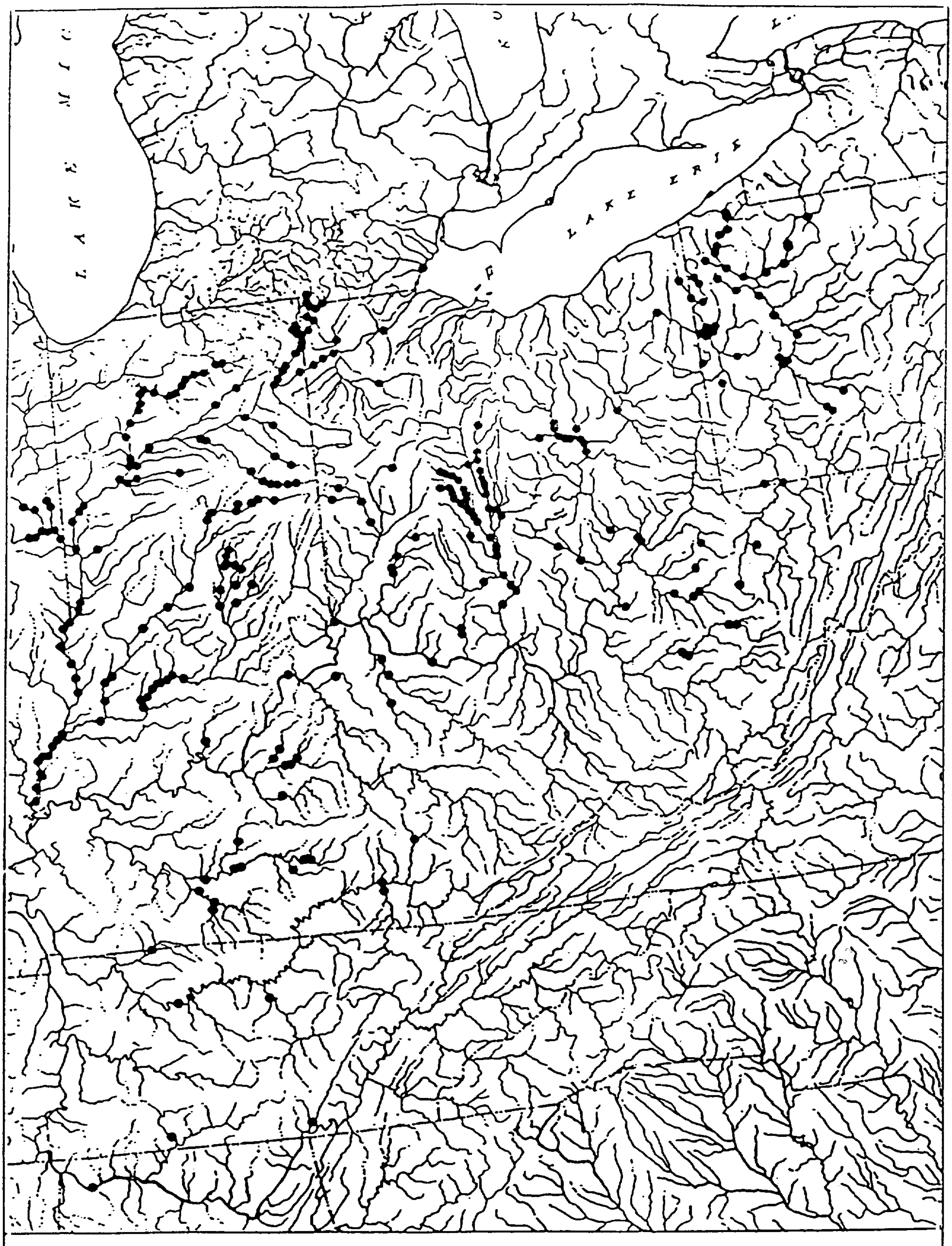


FIGURE 3. Clubshell - historical range.

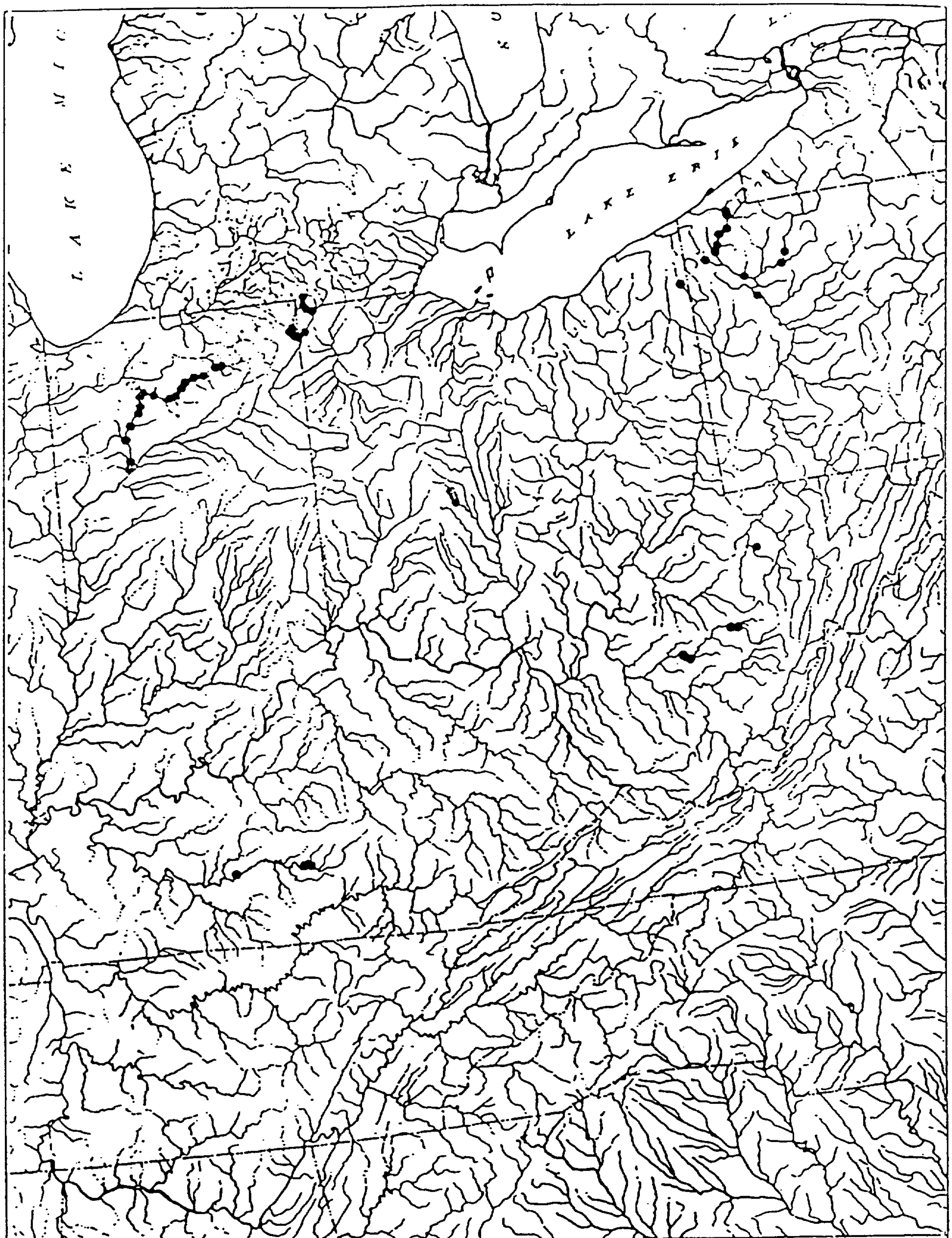


Figure 4. Clubshell - presumed present range.

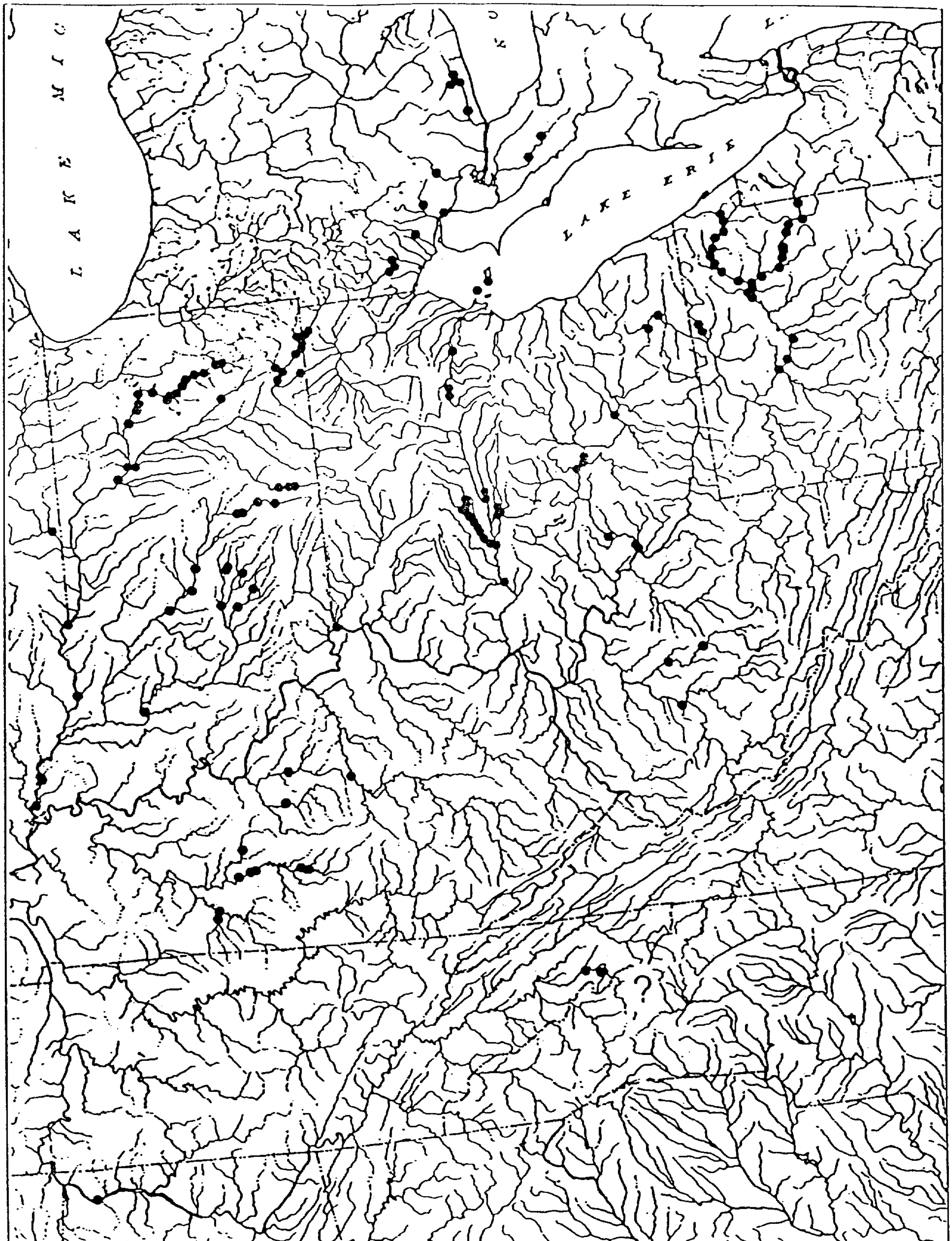


Figure 5. Northern Riffleshell - historical range.

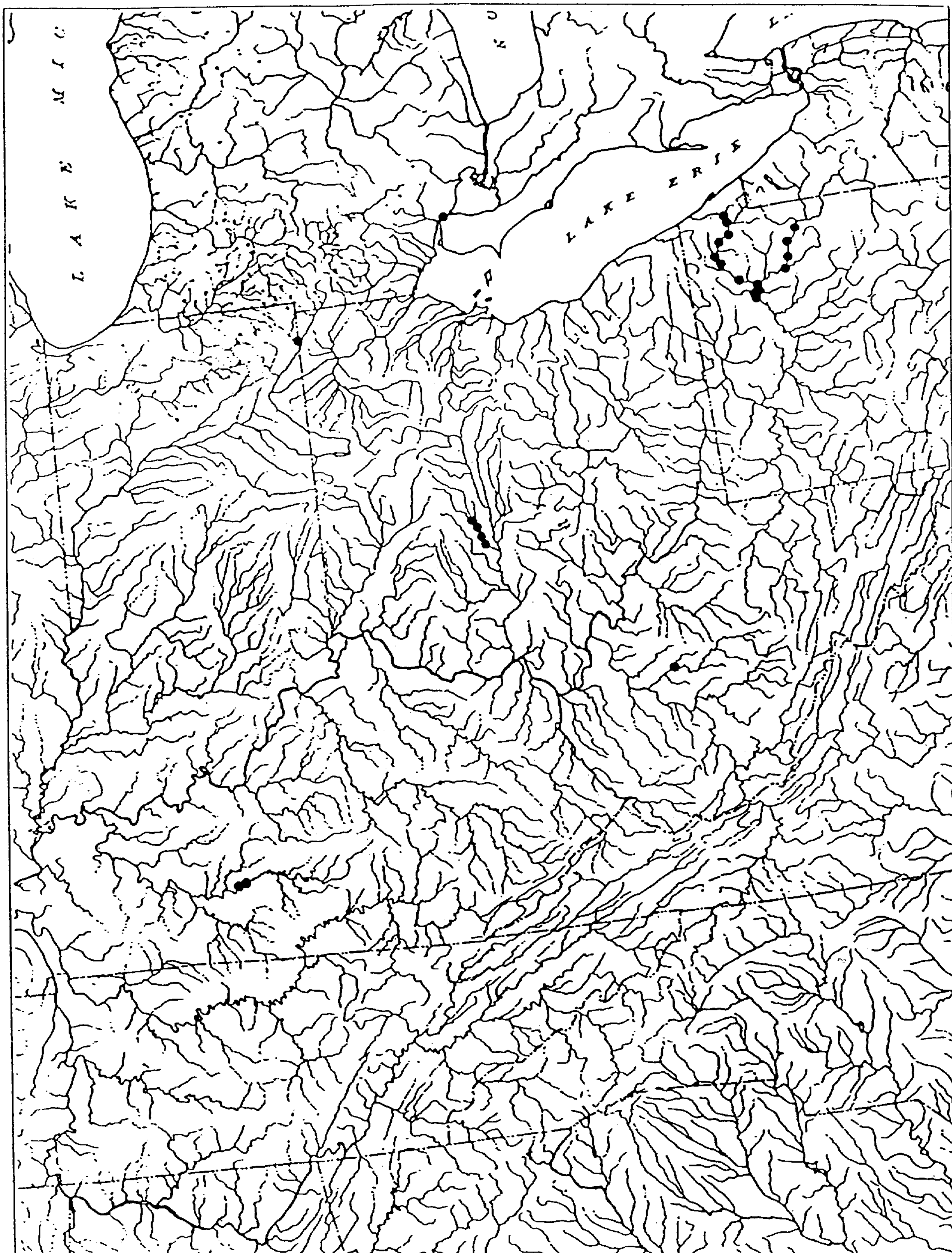


Figure 6. Northern Riffleshell - presumed present range.

Table 1. Historical (H) and present (P) occurences of the clubshell and northern riffleshell.

Drainage	General Locality	State/Province	clubshell	riffleshell
Lake Erie	La Plaisance Bay	MI	H	
	Bass Islands	OH		H
	Pelee Island	Ontario		H
Sydenham River	Sydenham River	Ontario		H
River Raisin	River Raisin	MI		H
	Macon Creek	MI		H
Huron River	Huron River	MI		H
Detroit River	Detroit River	MI/Ontario		P
River Rouge	Upper Rouge	MI		H
Clinton River	Clinton River	MI		H
	NB Clinton River	MI		H
Black River	Black River	MI		H (?)
Sandusky River	Sandusky River	OH		H
Maumee River	Maumee River	IN, OH	H	H (IN)
	Blanchard River	OH	H	
	Tiffin River	OH	H	
	St. Marys River	IN	H	H
	St. Joseph River	IN, MI, OH	H	H (IN, OH)
	Nettle Creek	OH	H	
	West Branch	MI, OH	P	
	Fish Creek	IN, OH	P	P
Wabash River	Wabash River	IL, IN	H	H
	White River	IN	H	H
	West Fork White	IN	H	H
	Big Killbuck Creek	IN	H	
	East Fork White	IN	H	H
	Big Blue River	IN	H	H
	Flat Rock River	IN	H	H
	Conns Creek	IN	H	H
	Brandywine Creek	IN	H	H
	Sugar Creek	IN	H	
	Vermillion River	IL	H	H
	North F Vermillion	IL	H	
	Salt Fork Vermillion	IL	H	
	Middle F Vermillion	IL	H	
	Wildcat Creek	IN	H	H
	NF Wildcat Creek	IN	H	

Table 1. (cont.)

Drainage	General Locality	State/Province	clubshell	riffleshell
Wabash R. (cont.)	Tippecanoe River	IN	P	H
	Eel River	IN	H	H
	Mississinewa River	IN	H	H
	Salamonie River	IN	H	
Ohio River	Ohio River	IN, KY, OH, WV	H	H (KY, OH, WV)
Blue River	Blue River	IN	H	
Tennessee River	Tennessee River	AL	H	H
	Elk River	TN	H	
	Sequatchie River	TN	H	
	Nolichucky River	TN		H (?)
Cumberland River	Cumberland River	TN	H	
	Red River	TN	H	
	Stones River	TN	H	
	Big South Fork	TN	H	
	Drakes Creek	TN	H	
	Rockcastle River	KY	H	
Green River	Green River	KY	P	P
	Russell Creek	KY	H	
	Barren River	KY	H	H
	Drakes Creek	KY	H	H
	Nolin River	KY	H	H
Salt River	Salt River	KY	H	H
	Brashears Creek	KY	H	
	Beech Fork	KY	H	H
	Floyds Fork	KY	H	
Kentucky River	Kentucky River	KY		H
	Eagle Creek	KY	H	
Great Miami River	Great Miami River	OH	H	
	Stillwater River	OH	H	
Licking River	Licking River	KY	H	
	South Fork	KY	H	
Little Miami River	East Fork	OH	H	
Ohio Brush Creek	Ohio Brush Creek	OH	H	
Scioto River	Scioto River	OH	H	H
	Paint Creek	OH	H	
	Deer Creek	OH	H	
	Big Darby Creek	OH	H	P

Table 1. (cont.)

Drainage	General Locality	State/Province	clubshell	riffleshell
Scioto River (cont.)	Little Darby Creek	OH	P	H
	Treacle Creek	OH	H	
	Little Walnut Creek	OH	H	
	Big Walnut Creek	OH	H	H
	Alum Creek	OH		H
	Olentangy River	OH	H	H
Kanawha River	Kanawha River	WV		H
	Elk River	WV	P	P
Middle Island Creek	Middle Island Creek	WV	H	
Hocking River	Hocking River	OH	H	
Little Kanawha River	Little Kanawha River	WV	H	
	North Fork Hughes	WV	H	
	North Fork Hughes	WV	H	
Muskingum River	Muskingum River	OH	H	H
	Tuscarawas River	OH	H	H
	Walhonding R.	OH	H	
	Killbuck Creek	OH	H	
	Mohican River	OH	H	
Hughes River	North Fork	WV	H	
	South Fork	WV	H	
Allegheny River	Allegheny River	PA	P	P
Monongahela River	West Fork	WV	H	
	Cheat River	PA	H	
	Dunkard Creek	PA	H	
	Hackers Creek	WV	P	
Ohio River	NF Little Beaver	PA	H	
	Raccoon Creek	PA	H	
	Beaver River	PA	H	
	Connoquenessing Cr.	PA	H	
	Shenango River	PA	H	
	Mahoning River	OH, PA	H	H
	W Branch Mahoning	OH	H	
	Neshannock Creek	PA	H	
	Pymatuning Creek	OH, PA	H(PA), P(OH)	
Allegheny River	Conemaugh River	PA	H	
	Loyalhanna Creek	PA	H	
	Buffalo Creek	PA	H	

Table 1. (cont.)

Drainage	General Locality	State/Province	clubshell	riffleshell
Allegheny R. (cont.)	Sandy Creek	PA	H	
	French Creek	PA	P	P
	Conneaut Outlet	PA	P	
	Conneauttee Creek	PA	P	
	LeBoeuf Creek	PA	P	P
	Conewango Creek	PA		H

over 150 miles. Freshdead individuals were found at an additional ten sites. In all, living or freshdead specimens were found in 63% of the sites studied, although weathered shells occurred at 97% of the sites. The ages of individuals ranged from three to 17 years, indicating that this population probably is reproducing. Muskrat predation seemed to be a major cause of death at many sites, based on numerous shells in middens. The extent of this predation and its importance needs to be rigorously investigated.

The clubshell was found as weathered shells at eight sites, and as "fairly fresh" at a ninth in the Mississinewa River in Indiana (H. Dunn, Ecological Specialists, Inc., St. Peters, Missouri, *in litt.* 4 January 1994). That group also recorded weathered and subfossil shells from two sites in the Salamonie River, Indiana. So far, there is no evidence that the clubshell still lives in those drainages. Additional surveys will be conducted in 1994.

The Mahoning River (Pennsylvania, Ohio) has historical records and may still harbor populations. These areas need immediate survey work. In 1993, ten live clubshells were found in four sites in Pymatuning Creek in Ashtabula County, Ohio (Huehner and Corr 1994).

The clubshell was once widely distributed in the Maumee River system (Clark and Wilson 1912). Although now a part of the St. Lawrence River system, at the end of the Wisconsian glacial stage the Maumee River flowed southwest into the Wabash River. Many Ohio River system species found in the Maumee drainage may date from this time (Walker

1913, Stansbery 1961). Species also may have crossed between the Wabash and Maumee Rivers via the Wabash and Erie Canal. In fact, Goodrich (1914) reported the clubshell from this canal. Despite their method of entry, only two tributaries of the St. Joseph River of the Maumee River now have populations of the clubshell. Both are small, cool, shallow creeks. Living specimens are rare in Fish Creek (Indiana and Ohio) for a distance of approximately 20 miles (Hoggarth 1987, Watters 1988b). In a survey of the lower seven miles of Fish Creek in late 1993 and early 1994, no living specimens were found, although freshdead shells were not uncommon. The East Fork West Branch St. Joseph River in Ohio and Michigan also has a population, particularly in a ten-mile long area in Hillsdale County, Michigan (Watters 1988b). This population has been in existence for many years (Winslow 1918, Strayer 1979), and many individuals are in excess of 12 years old.

The Elk River, Kanawha River drainage, in West Virginia has a population of the clubshell between Sutton Dam and Sycamore Creek in Braxton and Clay Counties (J. Clayton, West Virginia Department of Natural Resources, pers. comm. 6 August 1993). This population was discovered in the 1960s. More research work is needed to determine the status of this species in the Elk River.

An August 1993 survey of Hackers Creek of West Fork River, Monongahela River drainage, in West Virginia found many living individuals in approximately a 100-yard reach (J. Clayton, WVDNR, pers. comm. 6 August 1993). Similar isolated populations probably remain to be discovered elsewhere.

The clubshell has been found in small numbers as freshdead shells in the Green River of Kentucky in Hart and Taylor Counties (R. Cicerello, Kentucky State Nature Preserves Commission, pers. comm. 9 August 1993; R. McCance, Kentucky State Nature Preserves Commission, *in litt.*, 9 November 1993). It is not known if there are reproducing individuals. This area will be more thoroughly investigated. There are historic records of the clubshell for the Nolin and Barren Rivers, but it is apparently extirpated.

Once common throughout the Big Darby Creek drainage, today the clubshell only exists in small numbers in a twelve mile long reach of Little Darby Creek in Madison County, Ohio (Watters 1986, 1990, in press). Although apparently reproducing, the population is in danger from various outside sources. The clubshell has not been found living in Big Darby Creek proper since 1971 (OSUMZ records), but the removal of a low head dam in 1990 at the mouth of Little Darby Creek may allow this species to reclaim its original distribution in Big Darby.

Populations, some considered large, now exist in the Allegheny River and tributaries. LeBoeuf Creek in particular, below Lake LeBoeuf to its mouth at French Creek, has a dense, apparently reproducing population. Additionally, French Creek, for about a mile, below this confluence also supports numerous clubshells. Conneaut Outlet (tributary of French Creek) has a small sparse population (C. Bier, Western Pennsylvania Conservancy, pers. comm. 24 August 1993). The Allegheny River proper supports what appears to be a sparse viable population, but with low numbers and a discontinuous distribution over 66+ miles (C. Bier, WPAC, *in litt.* 6 January 1994).

EPIOBLASMA TORULOSA RANGIANA

The northern riffleshell has a distribution similar to that of the clubshell, except that it ranged farther north into Michigan and Ontario in tributaries of Lake Erie, Lake St. Clair, and the Detroit and St. Clair Rivers. One of these populations, in the Black River, has been largely extirpated by recent channelization activities. Prior to this construction, 115 individuals were transplanted to cages placed in the Detroit River (M. Rabe, Michigan Natural Features Inventory, pers. comm. 6 April 1993), where they remain at this time. Individuals (110) in the Detroit River also were relocated based upon the opinion that these individuals would be eliminated by zebra mussels in 1993. Some of these were taken upstream toward the St. Clair River (T. Weise, Michigan Department of Natural Resources, pers. comm. 26 August 1993). Another 23 were placed in the Belle Isle Aquarium on 10 October 1992 for life history studies, but these have now died of uncertain causes (J. Anderson and D. Sweet, Belle Isle Aquarium, pers. comm. 27 August 1993).

The northern riffleshell was once locally common in the Sydenham River of Ontario (Clarke 1973, 1978). Recent collections from this river have failed to find any living specimens, and it is considered extirpated (Mackie and Topping 1988), although fairly fresh shells have been found in the past decade by private collectors.

Populations of this species in the Allegheny River and French Creek are the largest remaining. It is abundant in several reaches of French Creek, where hundreds of specimens may be found in middens in a short distance (pers. obs.). It was found in LeBouef Creek only as dead valves (C. Bier, WPAC, pers. comm. 24 August 1993). In the Allegheny River the populations range from viable to those with apparently depressed vigor, with an overall known broken distribution of some 80 miles (C. Bier, WPAC, *in litt.* 6 January 1994).

Ahlstrom (1930: 44) once remarked that at the Bass Islands, northern riffleshell "was everywhere, but not common." Living individuals from western Lake Erie (Bass Islands) were last seen in the 1960's and this species is presumed extirpated from there, particularly considering introduction of the zebra mussel.

Although once widely distributed in the Wabash, Muskingum, and Sandusky River drainages, there is no evidence that any populations remain. The Mahoning and Little Mahoning Rivers (Pennsylvania, Ohio) have historical records and may still harbor populations. These areas need immediate survey work.

The northern riffleshell was found as weathered shells at four sites in the Mississinewa River in Indiana (H. Dunn, ESI, *in litt.* 4 January 1994). Additional surveys will be conducted in 1994. Populations in the Tippecanoe River and Sugar Creek (East Fork White River) in Indiana appear to have been recently extirpated, although some may still exist in low numbers (R. Anderson, INDNR, *in litt.* 15 December 1993).

This species was recently found living in the Elk River of West Virginia in Clay County (ESI 1993b). Additional survey work is needed to determine the extent of this population. Two other Federally endangered mussels have recently been found in this river as well: *Pleurobema clava* and *Lampsilis abrupta*.

Freshdead shells of the northern riffleshell have been found in the Green River in Hart and Edmonson County, Kentucky. It is not known if a reproducing population exists. Additional work will be conducted (R. Cicerello, KYNPC, pers. comm. 9 August 1993; R. McCance, KYNPC, *in litt.* 9 November 1993).

Living and freshdead individuals of this species were very rare in Fish Creek (Maumee River drainage, Ohio) in 1988 (Watters 1988b). Other examples have not been found in the most recent surveys (R. Anderson, INDNR, pers. comm. 5 May 1993; pers. obs. 1994).

Once common in Big Darby Creek (Ohio), this species is now represented by a small population in a 15-20 mile stretch in Pickaway and Franklin Counties (Watters 1986, 1990, in press). Fewer individuals were found in a 1990 survey than in 1986. It is known from a few old shells from Little Darby Creek, but apparently was never common there. The removal of a dam at the mouth of Little Darby now may enable the northern riffleshell to extend its range there, where water quality and habitat generally are equal to or better than in Big Darby.

BIOLOGY

The clubshell typically burrows completely beneath the substrate, apparently relying on water to percolate between the sediment particles (Watters 1990). Consequently, the species is very susceptible to siltation, which clogs the substrate interstices and suffocates the animal. The clubshell generally is found in clean, coarse sand and gravel in runs, often just downstream of a riffle. It cannot tolerate mud or slackwater conditions.

The riffleshell also occurs in packed sand and gravel in riffles and runs (Watters 1990). Its existence in the western basin of Lake Erie apparently was due to sufficient wave action producing continuously moving water. The species buries itself to the posterior margin of the shell, although females may be more exposed, especially during breeding season.

Although more specific biology of the riffleshell and the clubshell largely is unknown, the following general account of unionid biology is applicable.

Freshwater mussels are filter-feeding, essentially immotile animals. Oxygen and food are acquired across an extensive gill surface, and metabolic waste is released into the surrounding water. Because North American species lack true siphons, or tubes for water intake and release, most species are confined to burrowing only to the posterior edge of the shell. This renders them susceptible to predators, desiccation, and temperature and other environmental extremes. Nevertheless, many species live for 20-30 years, and some up to 140 years (Bauer 1987c).

Unionid food has been the subject of debate. Allen (1914) found the gut to contain mostly diatoms and "other algae," and in 1921 suggested that mussels feed on bacteria, protozoans, and organic particles. Churchill and Lewis (1924) agreed with Allen, finding that diatoms passed through the digestive system intact. Fikes (1972) maintained *Amblema plicata* for five months using an alga as food, but Imlay and Paige (1972) suggested that mussels fed on bacteria, protozoans, and the by-products of other food (such as fish food), rather than on the food itself. Bisbee (1984) found different proportions of algal species in the guts of two mussel species, suggesting that unionids selectively feed. Yeager *et al.* (1993) believed food for juveniles consisted of interstitial bacteria, yet an algal mix plus silt was suggested as food by Humphrey and Simpson (1985) and Gatenby *et al.* (1993). Small amounts of silt have been found to enhance survivorship in cultured mussels (Humphrey 1987b, Hudson and Isom 1984, Hove and Neves 1991) for undetermined reasons. Efforts to maintain the northern riffleshell at the Belle Isle Aquarium on dried *Chorella*, *Spirulina*, and yeast combinations were unsuccessful (J. Anderson and D. Sweet pers. comm. 27 August 1993). Obviously more research is needed to identify the food items of unionids.

Gametogenesis for North American unionaceans is initiated by changes in water temperature. Generally there is only one breeding season a year, although *Glebula rotundata* has been shown to breed multiple times a year (Parker *et al.* 1979, 1984), and *Cumberlandia monodonta* may breed twice a year (Howard 1915, Gordon and Smith 1990). Abnormally low water temperatures may delay reproduction. Constant low water temperatures, such as found below some dams, may prevent reproduction from ever taking place.

Typically sexes are separate, although small numbers of hermaphrodites have been found in most populations of many species (Poupart 1706; Fischerstrom 1761; van der Schalie 1966, 1970; Heard 1979), but have not been detected as yet in the clubshell or riffleshell. The number of hermaphrodites may increase in low population densities (Kat 1983, Bauer 1987c), and thus be an important consideration in managing rare species. Females move unfertilized eggs into specialized regions of the gills, called marsupia. The eggs are held there in water tubes, which may morphologically change during breeding (Smith 1979, Kays *et al.* 1990, Richard *et al.* 1991). The marsupial region of the gill during breeding either does not function as a site of respiration (Richard *et al.* 1991), or operates at greatly reduced efficiency (Allen 1921, Tankersley and Dimock 1992). The marsupial region may remain non-respiratory during the non-breeding season as well (Richard *et al.* 1991).

Males liberate sperm into the water, sometimes as spherical aggregates (Utterback 1931, Lynn 1987). The sperm of the clubshell and riffleshell are unknown, but may also occur in aggregates. Females downstream take up the sperm with incoming water. Eggs are fertilized in the water tubes. Fertilization success may be related to population density, with a threshold density required for any reproductive success to occur (Downing *et al.* 1993). The developing embryos are physiologically isolated from the outside medium (Kays *et al.* 1990, Gardiner *et al.* 1991). Larval shells are formed from concretions in the gills that act as a source of calcium carbonate (Silverman *et al.* 1985). Minute bivalved larvae, or glochidia, develop over a period of days to months. Unionaceans are divided into two behavioral groups based upon the duration that glochidia are held in the marsupia. Bradytictic or long-term breeders hold these larvae until the following spring or summer. Tachytictic or short-term breeders will release them later the same year, usually by July or August.

As larvae, unionids in North America parasitize a vertebrate host. With one known exception, that host is a fish. Although claims have been made that several species may complete their metamorphosis without a host (Lefevre and Curtis 1911, Howard 1914b), most evidence suggests that unionids are obligate parasites. The degree of host specificity has been debated, but too little is known of these host-parasite relationships to access to what degree a species is specific. Many early host identifications were made haphazardly and cannot be relied upon. Recent work suggests some degree of host specificity, with the unionid using ecologically and phylogenetically interrelated hosts. However, much work remains to be done on this association. The hosts for most unionid species are unknown.

Hosts are infested with glochidia when they come into contact with them in the water or on the substrate. Different unionid species have different methods of releasing larvae. Some simply expel the glochidia out their exhalent siphon along with water and waste products. Hosts either take in suspended glochidia and pass them over their gills, where they attach, or they contact larvae on the substrate, where the parasites attach to the fins or skin. Other unionids bind numbers of glochidia into long mucus matrices called conglutinates (Chamberlain 1934, Fuller 1971). These may be colored and resemble worms or other food items. The host's gills are infested when the conglutinates are eaten.

Once shed by the female, glochidia must acquire a suitable host or die, usually within 24 hours. Liberated glochidia may travel miles downstream in currents (Clark and Stein 1921). Estimated chances of a glochidium surviving to transform and excyst range from 0.0001% (Jansen and Hanson 1991) to 0.000001% (Young and Williams 1984). Although some species may compensate for this with high fecundity over many years (Bauer 1987b), other species have been shown to develop late and then reach early senescence (Downing *et al.* 1993). Because of the manner in which hosts acquire glochidia, it is not surprising to find that glochidia are overdispersed, that is, most hosts are either unparasitized, or carry but a few glochidia, whereas a very small number of hosts bear most of the parasite burden (Bangham 1940, 1955; Weir 1977; Dartnall and Walkey 1979; Neves and Widlak 1988). However, even heavily infested hosts show little ill effects, with few exceptions.

The glochidia clamp down on the host tissue, and cause cells to lyse. This fluid forms part of the food for the developing parasite (Arey 1924b, 1932b; Blystad 1924). Minute pores in the glochidial shells are thought to facilitate uptake of host nutrients (Rand and Wiles 1982, Kwon *et al.* 1993, Jeong *et al.* 1993). A host wound reaction forms a "cyst" around the glochidium (Faussek 1895; Arey 1921, 1932a). During growth, the larva will resorb much of its own tissue, including the adductor muscle and much of the mantle (Young 1911; Blystad 1924). After a certain amount of time (from hours to weeks), depending on water temperature (Schierholz 1889, Howard and Anson 1923, but see Young 1911) and individual species, the glochidium transforms to a juvenile and excysts. The juvenile burrows into the substrate (Bauer 1986, Clarke 1986, Buddensiek *et al.* 1993) or attaches to a larger object with a byssal thread. The thread is lost in adults.

Potential hosts may possess one of two types of immunity to attached glochidia. Natural immunity occurs in unsuitable hosts, which have immunological defenses against the glochidia (Howard 1914a, Bauer and Vogel 1987). Acquired immunity occurs when a suitable host has been previously parasitized, and has built up a temporary immunity. The number of exposures needed to achieve acquired immunity depends on the degree of prior infestations and duration between them (Lefevre and Curtis 1910, Surber 1913, Reuling 1919, Arey 1924a, Bauer 1987a). Therefore, juvenile hosts may be the most susceptible to parasitization (Bauer 1987a, Jansen and Hanson 1991, but see Young 1911 and Young *et al.* 1987). Acquired immunity to one unionid species may give the host immunity to others (Reuling 1919). In both natural and acquired immunity, encysted glochidia are killed. The tissue may be sloughed off (Arey 1932c, Fustish and Millemann 1978, Zale and Neves 1982, Waller and Mitchell 1989) or persist. Acquired immunity may be lost if no subsequent reinfestation occurs within a certain time period, and the fish may become susceptible to parasitization again. However, the amount of time needed to lose acquired immunity is not precisely known.

This host-parasite relationship apparently arose as a means of dispersal for the unionids. Lacking great motility and internal fertilization, unionids would be doomed to be carried to the sea over many generations. By attaching themselves to a highly motile host, such as a fish, they are dispersed within and between drainages. Records of dispersal on the feet of waterfowl largely are apocryphal (Rees 1965).

The hosts for *Pleurobema clava* are unknown. The closely related Cumberlandian *Pleurobema oviforme* has been shown to use central stoneroller, common shiner, fantail darter, river chub, and whitetail shiner (Neves 1983, Weaver *et al.* 1991). It was gravid from May through July. Related fishes in the range of the clubshell may be suitable hosts. Ortmann (1919) reported that *clava* was found gravid from May to July, and was tachytictic.

The clubshell typically lives completely buried in the substrate. It is possible that it comes to the surface during the breeding period. This behavior has been seen in other unionids (S. Ahlstedt, TVA, pers. comm. 13 October 1992), but has not been observed in the clubshell. Females are not known to have any particular displays or behaviors to lure fishes.

The hosts of *Epioblasma torulosa rangiana* also are unknown, but the similar Cumberlandian *Epioblasma capsaeformis* uses banded sculpin, and dusky, redline, and spotted darters (Yeager 1986). It was gravid from April through July. *Epioblasma brevidens* uses banded sculpin, greenside darter, logperch, redline darter, snubnose darter, and spotted darter (Yeager 1986). *Epioblasma florentina curtisi* parasitizes rainbow darter (Buchanan 1987), and *Epioblasma triquetra* uses banded sculpin and logperch (Yeager 1986, Sherman 1993). Again, similar species in the range of the northern riffleshell probably act as hosts. Ortmann (1919) reported that *rangiana* was gravid in September, and probably August, and was bradytictic.

The riffleshell during breeding has a spongy, pure white mantle lining that is displayed by the female. This can be seen from several yards away in clear water (pers. obs.), and may function to lure hosts. No mantle flap undulations or other behaviors have been noted.

Natural predators of metamorphosed mussels consist of fishes, birds, muskrats, and raccoons. In Europe, the hooded crow has been shown to drop mussels from the air to crack them open (Berrow 1991). Baker (1918) listed sheepshead, lake sturgeon, spotted sucker, common red-horse, and pumpkinseed and others as fish predators. Hanson *et al.* (1989) and Convey *et al.* (1989) have reported muskrats eating up to 37,000 mussels in a year in an Alberta lake. This may be an important source of predation on endangered species in some areas (Neves and Odom 1989). Both the clubshell and riffleshell are common in middens in Pennsylvania sites (C. Bier, WPAC, *in litt.* 6 January 1994).

Unionids often are parasitized by unionicolid mites (Mitchell 1965, Davids 1973) and monogenic trematodes (see review of Hendrix *et al.* 1985), which feed on gill and mantle tissue. Chironomid larvae may consume up to 50% of the mussel gill (Gordon *et al.* 1978), interfering with respiration and reproduction. Observed symbiotic relationships with ciliates may not be parasitic (Antipa and Small 1971). Leeches also may infest unionids.

REASONS FOR DECLINE AND THREATS TO CONTINUED EXISTENCE

Few mussel species have declined in numbers as drastically as have these two species. The clubshell in particular was once widespread and common. The decline of these two species undoubtedly is not due to any one cause, but to several compounding problems. For instance, potential threats to populations in the Green River of Kentucky include cool water discharge from dams, runoff from agricultural areas, and untreated wastewater and industrial effluents. Major point discharges in this area include Campbellsville, Greensburg, Munfordville, and Caveland Sanitation System, and Little Pitman Creek (R. Cicerello, KYNPC, *in litt.* 2 April 1993). As another example, perennial threats to the clubshell in Little Darby Creek (Ohio) population of urbanization, impoundment, land fills, etc., could easily extirpate this species from the system. Pollutants, silt, and sediments in runoff from agricultural and nonpoint urban sources are recognized as current and future threats to this population.

The fact that the clubshell and northern riffleshell have both very similar historic and recent ranges suggests that they have declined for similar reasons. Some of these reasons are discussed in general terms below.

SILTATION

As filter-feeders on microscopic food items, freshwater mussels are very susceptible to smothering by silt and other sediments in the water (Ellis 1936), and to pollutants carried by these sediments. Siltation also may result in reduced dissolved oxygen and increased organic material at the substrate level (Ellis 1936, Harman 1974). Consequently, agricultural, construction, and forestry runoff may be important causes of death in mussels. At sublethal levels, silt interferes with feeding and metabolism in general (Aldridge *et al.* 1987). Susceptibility to silt differs from species to species (Marking and Bills 1980). All the river systems in which these species now exist are susceptible to runoff.

IMPOUNDMENT

Impoundment drastically changes the biotic makeup of the impounded region, as well as the area immediately downstream (Ellis 1942, Neel 1963, Stansbery 1973, Ridley and Steel 1975, Baxter 1977, Buddensiek *et al.* 1993, Parmalee and Hughes 1993). Species and their hosts that require oxygenated, fast-flowing water quickly are eliminated. This includes most of the presently endangered mussel species, and nearly all of those that have become extinct. Most mussel species normally occur in shallow water, not in impoundment depths (Salmon and Green 1983, Haag and Thorp 1991).

Impoundment reduces the growth and reproductive effort of mussels. Individuals in cold, deep water grow slower than those in warmer water (Evermann and Clark 1920, Harman 1974, Hanson *et al.* 1988). Continuously cold water may prevent mussels from reproducing.

Impoundment also leads to an increased silt load by reducing water's capacity to carry sediments (Bates 1962, Negus 1966). The eutrophication that often accompanies impoundment has been suggested as a major source of mortality in mussels (Bauer 1988). Changes in the fish fauna, and therefore the availability of hosts, also occur with impoundment (Williams *et al.* 1992).

Dams represent distributional barriers to fish hosts, and therefore to the mussels themselves. The zoogeographic patterns of several species suggest a dam-limited range (pers. obs.). Dams also act as sediment traps, often having many feet of silt and debris caught on their upstream side. These areas generally are without mussels. The tailwaters on the other hand often have dense beds. This is mistakenly believed by many to be a benefit of the dam. Actually, these beds represent the last remaining portions of the river *in general* prior to impoundment. The tailwaters are the only areas left that still have oxygenated, fast moving water. This is exemplified by the distribution of beds in the lower Muskingum River, Ohio (Stansbery and King 1983, ESI 1993c).

IN-STREAM SAND AND GRAVEL MINING

Mining "river gravel," beyond what is needed to maintain navigation channels, may be devastating on mussels. Many are physically removed with the substrate, others are buried or crushed. Additionally, sediments and silt are released that may affect downstream populations. Head-cutting, where the upstream border of the pit collapses and moves upstream, also may be a serious consequence of sand and gravel operations.

POLLUTANTS

Havlik and Marking (1987) give a review of the effects of contaminants on mussels. Readers are urged to consult this source for more information on this section. Some particular effects are given here.

Much of the remaining range of these two species are in agricultural land subjected to pesticide and fertilizer runoff. Although effects of pesticides are species-specific, in general sub-lethal levels of PCB's, DDT, Malathion, Rotenone, and other compounds inhibit respiratory efficiency and accumulate in the tissues. Mussels were more sensitive to pesticides than many other animals tested. It is not known to what extent the clubshell and riffleshell are affected by pesticides, but these mussels undoubtedly are adversely affected to some degree by these pollutants. Research is needed to determine lethal and sub-lethal levels.

Mussels are particularly sensitive to heavy metals, more so than many other animals used in toxicological tests (Keller and Zam 1991). Responses may be species-specific (see example for copper in Jacobson *et al.* 1993). Behavioral responses may allow some adult mussels to survive short-term exposure (Keller 1993). Low levels of metals may interfere with the ability of glochidia to attach to the host (Huebner and Pynnönen 1992).

Glochidia were found to be very sensitive to ammonia from wastewater treatment plants, although much less so to septic systems, although the reasons for this observation are not clear (Goudraeu *et al.* 1993). At sub-lethal exposure, adult mussels exhibited decreased respiratory efficiency (Anderson *et al.* 1978).

Acidic water from mine runoff and sandy soils may eliminate mussels and preclude recolonization (Simmons and Reed 1973, Humphrey 1987a, Watters 1988a). During exposure to low pH, calcium for haemolymph buffering may be derived from the shell and mantle, but not from concretions in the gills used for glochidial shell construction (Pynnönen 1990). Mussels may be able to survive several weeks of exposure to relatively low pH because of this buffering (Mäkelä and Oikari 1992). However, low pH also interferes with the glochidia's ability to close its shells on a host (Huebner and Pynnönen 1992).

Liqouri and Insler (1985) gave circumstantial evidence that salinity was lethal to some glochidia. This may be a problem in runoff from salt used for clearing roads in winter throughout much of the range of both of these mussels. Salinity also is a concern near oil and gas production areas, such as the Green River.

The recent invasion of zebra and quagga mussels has compromised the continued presence of many mussel populations. Native mussels have been effectively eliminated from the western basin of Lake Erie by these exotics. Mortality may be caused by numerous factors including starvation, loss of fecundity, depleted oxygen availability, and beaching after storms. To date, outside of the St. Clair River, no zebra/quagga mussels have been found in areas of clubshell or riffleshell populations. Whether this will remain the case is unknown. Zebra mussels have been found in the Scioto River (the parent stream of the Big Darby system), and in one or more glacial lakes in the Tippecanoe River drainage.

CONSERVATION MEASURES

Several conservation measures are available to listed species pursuant to the Federal Endangered Species Act. Recognition through listing encourages and results in conservation actions by Federal, State, and private agencies, groups, and individuals. The Endangered Species Act provides for possible land acquisition in cooperation with the States and requires that recovery actions be carried out for all listed species. The protection required of Federal agencies and the prohibitions against certain activities affecting listed species are discussed below.

Section 7(a) of the Act, as amended, requires Federal agencies to evaluate their actions with respect to any species that is proposed or listed as Federally endangered or threatened. Regulations implementing this interagency cooperation provision of the Act are codified at 50 CFR Part 402. Section 7(a)(4) requires Federal agencies to confer informally with the Service on any action that is likely to jeopardize the continued existence of a proposed species or result in destruction or adverse modification of proposed critical habitat. If a species is listed subsequently, Section 7(a)(2) requires Federal agencies to ensure that any activities they authorize, fund, or carry out are not likely to jeopardize the continued existence of such a species or to destroy or adversely modify its critical habitat. If a Federal action may affect a listed species or its critical habitat, the responsible Federal agency must enter into formal consultation with the Service.

In addition to State Endangered Species Acts, some states, such as Ohio, have recent legislation making it illegal to possess over a certain number of mussels, even with a fishing license. Although neither species is of commercial value, they have been affected in the past by incidental take. Ohio and Indiana have now closed their waterways to all commercial collecting. In Pennsylvania, mussels are regulated as bait.

Several populations occur in mussel sanctuaries (Fish Creek, Ohio) or national parks (Green River, Kentucky), and presumably benefit from this association. The Green River in Mammoth Cave National Park also is a Kentucky Wild River, and other segments are a Kentucky Outstanding Resource Water. The Kentucky Division of Water is obligated to protect these habitats to sustain mussels. A portion of the population of riffleshell in Big Darby Creek is on property owned by The Nature Conservancy. Other populations are on property that has a watershed management plan (Fish Creek in Indiana, Michigan, and Ohio) or is the subject of local conservation groups (Big Darby Creek system, Ohio; portions of the

Tippecanoe River, Indiana; French Creek, Pennsylvania). To some extent, these sites are monitored for water quality and compatible land use.

An inventory of mussels in Mammoth Cave National Park was completed in 1990 through a cooperative effort of the National Park Service and the Kentucky State Nature Preserves Commission. Detailed monitoring of this area began in 1993. Water quality in the Green River is monitored by the Park Hydrologist, by the Kentucky Division of Water through the Wild Rivers Program, and by the U.S. Geological Survey. The Mammoth Cave Area Special Water Quality Project supports habitat protection by implementing agricultural "Best Management Practices," and the Caveland Sanitation Authority, which oversees construction and operation of new regional sewage treatment facilities. Public awareness of the mussels and the aquatic ecosystem is raised through interpretive programs for park visitors.

"Forest Watch" groups exist in the Allegheny, Hoosier, and Shawnee National Forests, which include parts of the historical range of both mussels. The Forest Service has promulgated regulations to ensure the protection of endangered species and their habitats (Forest Service Manual, Chapter 2670).

Mussels downstream from Green River Reservoir in Taylor County, Kentucky, historically were subjected to cold water discharge. Army Corps of Engineer personnel recently indicated that this discharge was changed a few years ago from "cold water" to "cool water." An attempt is made for these releases to follow historical water temperature curves. However, once a certain maximum temperature plateau range is reached, further increase does not occur. This is in part because of limited discharge capability from various reservoir levels (R. McCance, KYNPC, *in litt.* 9 November 1993).

L. White of Pennsylvania State University is studying these mussels and their hosts in French Creek, comparing DNA sequences in encysted glochidia with adult mussels to determine host identities. The author also is beginning a study of the hosts of these two species through artificial infestation.

RECOVERY STRATEGY

Effective recovery of the clubshell and northern riffleshell will necessitate a much better understanding of these species' biology and ecological requirements. However, because these species depend on riverine ecosystems, a broad initiative to conserve these dynamic systems can be undertaken concurrently with acquiring more detailed, species-specific information.

Conservation of the systems of which the clubshell and northern riffleshell are a part should begin immediately (where not already underway) with delineation of the ecosystems for both species and a comprehensive survey of the individual resources within each ecosystem. While the two mussels species, as well as other listed species such as *Lampsilis abrupta* that share the habitat, should be a primary focus of conservation strategies, the overall goal of these strategies will be to maximize the health of the system as a whole. These broad conservation plans can be then complemented (as needed) with measures, such as augmentation or translocation, for bolstering the population levels of each species.

Public awareness of the recovery needs for these species, particularly of the need for ecosystem protection, may be a decisive factor in the success of recovery efforts. Likewise, awareness on the part of technical specialists of the human concerns involved in broad conservation efforts may engender more confidence these efforts. An important component of this recovery process will be, therefore, to develop a two-way educational program for understanding the need for and means of achieving conservation of these species and their habitats.

PART II: RECOVERY

RECOVERY GOAL

The goal of this plan is to maintain and restore viable populations of the clubshell and northern riffleshell to a significant portion of their historic range by the year 2020.

RECOVERY OBJECTIVES

Objective 1. Reclassify populations of both clubshell and northern riffleshell from endangered status to threatened status when a significant proportion of the populations has been secured from foreseeable and controllable jeopardy. Reclassification will be considered when the following criterion is met:

A. Viable populations must be established in 10 separate drainages for each species. A viable population consists of sufficient numbers of reproducing individuals to maintain a stable or increasing population (see Task 1.44). These populations should include as many subpopulations as possible to maintain whatever fraction of the original genetic variability now remains. At this time, the following drainages for each species are identified as necessary for achieving recovery:

Clubshell	Northern riffleshell
Tippecanoe River, IN	Tippecanoe River, IN
East Fork West Branch St. Joseph River, MI, OH	Detroit River, MI, Ontario*
Fish Creek, IN, OH	Fish Creek, OH
Green River, KY	Green River, KY
Little Darby Creek, OH	Big Darby Creek, OH
Elk River, WV	Elk River, WV
French Creek, PA	French Creek, PA
Allegheny River, PA	Allegheny River, PA
plus two additional drainages	plus two additional drainages

* contingent on zebra mussel control

Objective 2. Remove the clubshell and northern riffleshell from the Federal list of endangered and threatened species when viable populations are established and protected through significant portions of both species' ranges. Delisting will be considered when the following additional criteria have been met:

B. For each species, each of the ten populations given in Criterion A must be large enough to survive a single adverse ecological event. Most populations at this time are localized and susceptible to such impacts. Therefore, the extent of most populations must be increased, either naturally or through translocation.

C. To this end, the populations and their drainages from Criteria A and B must be permanently protected from all foreseeable and controllable threats, both natural and anthropogenic.

Monitoring is required until the year 2020, the anticipated date of full recovery, or until viability of the species can be demonstrated. Evidence of recruitment and population stability is necessary to validate viability.

RECOVERY TASKS

1. Initiate and participate in ecosystem conservation efforts. All recovery activities for the clubshell and northern riffleshell should eventually feed into comprehensive ecosystem/watershed management programs. It is imperative that these efforts be conducted proactively in order to prevent further declines in these species' status. Activities should be designed to benefit the entire aquatic ecosystem of which the mussels are an important and particularly sensitive component (M. Holm, Mammoth Cave National Park, *in litt.* 22 December 1993).

   1.1 Delineate the ecosystems for the priority drainages identified for each species. Delineation of the mussels' ecosystems will involve identifying: (1) the extent of the river or creek stretch that supports a population, (2) upstream -- or downstream -- areas within which activities could have an effect on the population, and (3) riparian and upland areas associated with the river or creek within which activities could have an effect on the population.

1.2 Identify other resource values within these ecosystems. Possible host fishes, associated listed and candidate species, species diversity, other key animal and plant indicators, aquatic and riparian habitat elements, wild and scenic values, cultural values, and other important resources should be inventoried within each delineated ecosystem. Where such inventories are already underway, it should be ensured that the clubshell and northern riffleshell are addressed as integral ecosystem components.

1.3 Identify activities or practices within each ecosystem that may affect the clubshell and northern riffleshell as well as other sensitive resources. Activities and practices that may threaten these mussels (discussed in Reasons for Decline and Threats to Continued Existence), as well as activities that may benefit the mussels and other resources within each delineated ecosystem should be identified. These actions should then be assessed in regard to their potential effect and the time frame needed to alleviate these effects. As data gathered from Task 2.1 become available, they should be incorporated into these considerations.

1.3 As a near-term conservation measure, identify and participate in ongoing environmental planning and regulatory compliance processes within each ecosytem. Opportunities to become involved in reviewing and influencing state water quality standards, and to participate in resource assessment and land and water use planning processes (including watershed plans that are already underway), state river inventories, wild and scenic river designations, Habitat Conservation Plans, environmental review processes, permit reviews, etc., should be exploited. Relevant data from Task 2.1 should be made available for these planning and regulatory processes.

1.4 Help develop and implement comprehensive watershed plans. These plans should have as a primary objective the maintenance of the ecosystems on which these mussels and their hosts depend. To this end, decision-makers should be charged with adopting both short- and long-range goals and practices to ensure the prevention of any further decline of the species' status, and, where shown to be feasible, to rehabilitate habitat to the point where it can be used to support mussel populations in furtherance of recovery objectives. In addition, protection efforts directed toward other or multiple resources should be compatible with recovery of mussel species.

Because of the diverse nature of the drainages in which the populations of these two species are found, plans would have to be unique to each drainage. Such plans would include, but not be limited to, maintenance of minimum water flow and quality standards, acquisition of buffers and easements, alternate cultivation techniques, appropriate application of pesticides, and other in-stream and shoreline protection measures.

Each plan should be developed and implemented by both agencies and landowners. It is imperative that the public -- particularly landowners of both occupied and potential habitat -- be an active part of the planning and implementing of habitat management activities (see Recovery Strategy and Task 4).

Watershed management plans will recommend compatible and "best management" practices, identify incentives, and explain existing regulatory restrictions with the rightful expectation of enforcement. Full participation of local governments and constituencies is needed for successful implementation. These landscape-scale plans are the only realistic means of stabilizing or improving these mussels' populations.

2. Protect and manage mussel populations and their habitat on a site-specific basis. This task should be should undertaken by all agencies that have legal responsibility for protecting and recovering these two listed species. Recovery activities will carried out on Federal lands wherever possible, and include other listed and candidate species with similar ranges. However, few of the populations of the clubshell and northern riffleshell are within such areas (Green River in Mammoth Cave National Park, for example).

2.1 Monitor population status, including demographics, at existing sites through a collecting protocol. Additional surveys are needed to determine the existing population status of each species. Where applicable, surveys should be conducted in conjunction with ongoing activities, such as with the U.S. Forest Service in proposed timber harvest and road building units. Surveys should be conducted on a regular basis throughout the recovery period for these species, as stated under the Recovery Objectives.

To date, survey methods have varied widely among researchers, resulting in data that may not be comparable or repeatable. In order to more effectively characterize the demographics of a population, a standardized protocol for surveying and monitoring mussels should be developed, and its use mandated by agencies. Surveys should be conducted in a rigorous scientific method that adequately addresses the questions of population age structure. Suggestions for formal surveying of mussels may be found in Kovalak *et al.* (1986), Isom and Gooch (1986), Miller and Payne (1988, 1993), Downing and Downing (1992), and Miller *et al.* (1992).

2.2 Identify and map both actual and potential threats at existing sites and potential translocation sites. Water and sediment quality should be monitored in those drainages harboring either of the two species. In addition, the potential effects of proposed projects (commercial dredging, channelization, impoundment, etc.) on both existing populations and potential translocation sites (see Tasks and) should be identified and evaluated. Measures should be identified to eliminate existing threats and to address potential ones. This task is an important but dynamic concern.

For the most part, the threats to the drainages in which these mussels occur are known in only a generic way. Watershed or Conservation Plans (e.g., Habitat Conservation Plans) developed for each river system (under Task 1) should include identification of threats and site-specific measures to eliminate or minimize these threats to the extent possible.

2.3 Enforce all laws and regulations pertaining to the collection of specimens and protection of habitat. *Pleurobema clava* and *Epioblasma torulosa rangiana* are protected by the Federal Endangered Species Act of 1973, as amended, and under individual State endangered and threatened species laws and regulations, as well as laws specific to mussel protection (see Conservation Measures). The riverine habitat occupied these species receives some degree of protection under Federal and State water use and quality laws and regulations. As surveys are completed and new information is gathered, it should be provided to local, State, and Federal regulatory agencies for use during their permit review process.

2.4 Conduct site-specific protection and management programs. Key components of site-specific protection will include: (1) prevention of activities detrimental to maintenance of water quality standards, within specific areas of jurisdiction, (2) water quality monitoring, (3) review and input into water quality designations as needed to assure levels above that required by the mussel species and their hosts within a specific jurisdiction, and (4) monitoring of the effects of habitat management activities.

2.5 Conduct searches, as warranted, for additional populations. Additional populations of one or both of these species may exist, particularly in some portions of their known ranges. Searches of suitable habitat should be conducted in conjunction with other habitat protection activities.

3. Collect data on both species that are necessary for their recovery. In addition to monitoring the status of and threats to each population (Tasks 2.1 and 2.2), other baseline data are needed to provide a better scientific basis for conducting the inventory and planning efforts described in Task 1, for resolving site- and species-specific issues, and for potential mussel restoration efforts.

3.1 Determine contaminant sensitivity for each life stage. It has been shown that different life stages may differ in their sensitivity to contaminants. Glochidia and newly metamorphosed juveniles may be more susceptible than adults. Both sub-lethal and lethal levels of pesticides (including B.t.i., Dimilin, and molluscicides), discharges (including chlorines, ammonia, heavy metals, and pH and salinity changes), etc., should be determined.

3.2 Complete life history studies for each species. This information will be used in monitoring, conservation, and restoration efforts for both species.

3.21 Identify hosts for each species. At present, no hosts are known for either species, although related unionids have some hosts identified. Fishes that are closely related to those known hosts and are present in the drainages of the clubshell and riffleshell may be the best candidates. It also is important to determine breeding periods, and the availability of hosts in the area during these periods.

3.22 Determine life history, ecology, and status for identified hosts. Because these unionids are obligate parasites, it is important to know the ecology and behavior of their hosts. In particular, questions about whether the habitat can support the host as well as the mussel, whether the host is present in sufficient numbers at the right time for parasitization to occur, etc., must be investigated. Once hosts are determined, distribution and health of the host populations must be evaluated.

3.23 Identify nutritional requirements for each life stage. It is possible that the two species have different food requirements. Additionally, metamorphosed juveniles may feed on different items than adults. These requirements may be limiting factors for one or both species.

3.24 Determine stable population levels. Demographics of the largest extant populations should be used to identify a target population size and density. Only populations showing recruitment can be used for this task.

3.25 Estimate the impact of potential natural predators. Most of the remaining populations of these two species are sufficiently localized and sparse such that natural predators may constitute an important source of mortality. It may be necessary to reduce this pressure through management practices.

3.3 Characterize the habitat that best supports these species. This should include an analysis of results of water and sediment quality monitoring (Task 1.3), and should take an ecosystem approach.

3.31 Characterize historical sites. Using literature records, museum records, and archeological excavations, compare the historical composition of the site and its current state. This may help reveal why the species was extirpated in a given location, and spare other currently occupied sites the same fate. Translocation should not be undertaken if the cause of the original extirpation is not known or corrected.

3.32 Identify the constituent components of suitable habitat for each species. Information about currently occupied sites, including, for example, species composition, flow regime, water quality and chemistry, substrate characteristics, shoreline features, etc., will be used (1) as baseline data for monitoring threats and habitat management activities, and (2) to identify possible sites for translocations, based on similarity of habitat composition.

3.4 Undertake genetic studies to determine the species' limits. At present, there is no sufficient means to separate some populations of *Pleurobema clava* from *oviforme*. Should the pair be found conspecific, this would greatly increase the range and number of populations of this taxon and perhaps change its listing status. Likewise, the relationship between *rangiana* and *torulosa* needs to be understood. If *rangiana* is only the headwater expression of *torulosa*, then specimens of *rangiana* could be used to reintroduce *torulosa* to larger rivers.

3.5 Identify the potential effects of, and responses to, zebra and/or quagga mussel invasions, and their control measures. Although it is not known if these introduced mussels will invade populations of the clubshell and/or northern riffleshell, the likely effects of such an invasion should be determined. Whatever preventive or response measures to such an invasion might be called for should be initiated. This could include sequestering the clubshell and/or northern riffleshell in man-made refugia, transplanting whole populations to more remote areas, cryogenically preserving gametes and larvae, etc. Research that is being conducted into the spread and control of these invasive species should be followed and applied as appropriate to situations involving the ecosystems of the clubshell and/or northern riffleshell. In addition, many control measures for zebra mussels (chlorine, etc.) are likely to be detrimental to other molluscs, including the clubshell and riffleshell. The use of these nonspecific molluscicides must be carefully monitored and controlled, and the effects of these molluscicides should be determined for native mussels.

4. As needed, restore habitats and reintroduce the species to suitable areas. To meet the criteria for delisting of both species, either existing populations must increase in density and range, or new populations must be established. Translocation may thus entail: (1) bolstering a small population through artificial infestations or *in vitro* cultures from the same population, (2) bolstering a small population with individuals from a larger

one, and/or (3) introducing individuals to an area where they have been extirpated. In attempting to recover species in immediate jeopardy of extinction, all available methods to ensure survival must be considered; in this case, the remaining populations of these mussels are distant from one another, making interbreeding by natural methods a virtual impossibility. New populations will only be established by human intervention. In some cases habitat must be restored through land management practices such as establishing riparian corridors and buffers, removal of dams or restoring historic water temperatures in dam discharges, wise use of "dragging" stream beds for debris, limiting livestock access, etc. Wastewater treatment plants should be upgraded. Chlorination practices for these plants should be modified or abandoned in favor of less environmentally destructive methods.

Measures should be taken to minimize adverse effects on these two species. Point sources, including effluents from industry, wastewater or power plants, mines, etc., should be evaluated for their potential to adversely affect the mussels and the habitat. Appropriate actions must be taken to correct these problems. Watershed management plans should include tasks for monitoring of point sources, and for emergency responses to sudden, unforeseen environmental impacts.

4.1 Select potential translocation sites. Sites that may receive translocated mussels should be carefully chosen. Clearly, the site should have adequate substrate and water quality to promote survival. Translocation sites should not be in a location where the new population is subject to extirpation by a single, foreseeable and controllable event. Jenkinson and Heuer (1986) have outlined in detail their criteria for translocation sites. Summary criteria, including several not given by those authors, are given here.

- Historic sites. Literature and museum records often indicate sites that at one time harbored mussels, but from which the species is now extirpated. Often the cause of the loss is not precisely known. Extirpation may have been due to a transient event that killed the mussels or hosts, but from which no refugia were available to reintroduce the species. If habitat quality has recovered, such sites may be optimal choices for restoring the range of the species; however, such sites should be carefully investigated before mussel introductions take place. Translocation should not be made to historic sites without an understanding of the causes of the original extirpation.

- Presumed historic range. In many cases, streams or rivers were never surveyed before being degraded to the point where no evidence of the historical fauna remained. If within the historic range of the species, these systems should also be candidate translocation sites.

- Presence of the host. If the host(s) for a mussel is known, translocation sites should be selected based on the presence of that host(s). In lieu of such knowledge, the site should contain closely related or ecologically similar species to those known from currently occupied habitat.

- General health of transplanted mussels. Only healthy individuals should be used whenever possible. The potential to introduce new parasites or diseases to the relocation site should be realized. Dying or obviously diseased specimens should not be relocated.

- Other sites in drainages currently occupied by the species. Even if specific sites have no historical records for the species, such sites should be considered as a measure of last resort. This is particularly so if occupied sites are separated by dams or other obstacles to fishes that may have artificially isolated the population.

- Secure translocation sites. Translocation should not be made to sites which are currently unprotected, or in which threats to the species are known to exist.

- Artificial enclosures. If no other recourse is available, mussels may have to be moved to enclosures such as farm and hatchery ponds, and aquaria. Every effort should be made to maintain habitat parameters as closely as possible to those of wild populations, which may, admittedly, be difficult to do. Experiments with mussels kept in enclosures have produced mixed results, and generally have not dealt with rare species (Howard 1914c, 1916, 1922; Corwin 1920, 1921; Humphrey and Simpson 1985; Hanson *et al.* 1988; Humphrey 1988).

4.2 Translocate portions of existing populations. Translocation success varies from species to species and is dependent on handling. When possible, translocations should be conducted in a rigorous, scientific manner that will have heuristic value to future projects. To date, translocation success from one site to another has been ambiguous (Isley 1914, Clarke 1967, Imlay 1972, Ahlstedt 1979, Nelson 1982, Koch 1990).

4.21 Develop translocation protocol. Develop standard methods for translocating mussels in general that maximize survival. Protocol should include methods of transport (in water, on ice, etc.), transplantee placement in new site (random, aggregated), marking (tags, paint, etc.), and handling.

4.22 Translocate reproducing adults. This is the easiest method to release a species into a new area. It should be done after breeding season to improve the chances of moving gravid females. There may be a threshold density for reproduction (Downing *et al.* 1993) that may not be met in the new area without recruitment.

4.23 If needed, release metamorphosed juveniles. Metamorphosed juveniles also may be released, if there are not enough adults to risk translocation. This method may be the most efficient way to bolster endangered mussel populations. Metamorphosed juveniles may be obtained by one of two methods:

- Cultured *in vivo*. Hosts may be artificially infested in the laboratory and held until glochidia metamorphose and excyst. Currently, some metamorphosed juveniles may be held for several months.

- Cultured *in vitro*. Glochidia may be raised to metamorphosis without infesting a host on artificial media containing fish sera (Meyers *et al.* 1980; Hudson and Isom 1982, 1984; Isom 1983, 1986a, 1986b; Ison [sic] and Hudson 1984) or standard non-specific ingredients (Keller and Zam 1990; Johnson *et al.* 1993). Presently, glochidia of some species may be metamorphosed with 80-90% survivorship in non-specific media (A. Keller, USFWS, pers. comm. 26 August 1993).

4.24 Release infested fishes. It also is possible to infest artificially fishes and return them to the wild, where presumably the glochidia will metamorphose and excyst. Barney (1922) found circumstantial evidence that this process worked, but it has not received any attention since that time.

4.3 Release additional hosts. If existing hosts are identified, reproduction in extant populations theoretically may be bolstered by the release of additional host fishes (Bauer 1988). To the author's knowledge, this has not been tried.

4.4 Monitor new populations. Newly introduced populations through transplantation of adults may be easily monitored. These should be examined at least annually to determine mortality. Samples of substrate should be examined after 2-3 years for developing juveniles. Populations bolstered by release of infested fishes cannot be monitored in a rigorous way because of fish dispersal. Samples of presumed optimal habitat should be sampled every 2-3 years for juveniles.

5. Enlist public support for the recovery process through an outreach program and incentives. Public awareness is vital to achieving local and societal support for recovery of these species. Public support should be sought through traditional means such as educational and interpretive programs that can engage people's imagination and concern, as well as through innovative efforts to forge partnerships and provide incentives for thoughtful conservation of aquatic resources. The latter approaches can best be implemented through participation in local civic processes and through the landscape-scale plans discussed in Task 1.

5.1 Development and distribute an educational video. Most people are not aware of the role of mussels in nature, or their specialized life cycle. A video, either to be developed or modifying one that has already been produced, could be used to give people an appreciation of where mussels live, how they live, and why they should be protected.

5.2 Increase public and agency awareness of existing laws that protect these species. In addition to the Federal Endangered Species Act, most states have laws protecting mussels and other nongame wildlife. An explanation of these laws and their purpose should be provided to landowners and the general public (particularly during the watershed management planning process, Task 2.4) to demonstrate the value of mussels and the role that the public and their agency representatives play in protection of these species. State and Federal agencies should be encouraged to assist in the recovery process by vigorously enforcing all existing laws and regulations.

5.3 Encourage public involvement in the recovery process. Hands-on, field experience is the best way to generate interest and increase awareness of environmental issues. Agencies should promote campaigns such as "adopt a stream" cleanup programs, stream monitoring as classroom projects, and neighborhood watches for violators of clean water and wildlife laws (e.g., regulations against poaching and vandalism).

5.4 Develop and distribute educational materials. Brochures illustrating mussels and their life cycle should be made available through extension services, at parks and museums, and possibly through the mail. Additional brochures that focus on endangered or rare mussels should be developed and distributed.

5.5 Use media opportunities to reach the general public. The most efficient way to reach large numbers of people is through the media. Local newspapers and television newscasts may be receptive to stories about educational demonstrations, cleanup programs, and human interest stories involving mussels. Enthusiasm and thoughtfully worded explanations could go far in increasing public awareness of issues involving these species and their ecosystems.

5.6 Make presentations. Educational demonstrations of mussel natural history -- whether in the field or as a slide show -- could be given to a wide spectrum of audiences with a little customizing. Such demonstrations could be presented to scout troops, local conservation clubs, college seminars, agency representatives, etc. Lecturers should be able to speak to a wide audience and be prepared to answer complex questions. An accompanying hands-on synoptic collection would be very useful.

REFERENCES

Ahlstedt, S. 1979. Recent mollusk transplants into the North Fork Holston River in southeastern Virginia. *Bulletin of the American Malacological Union for 1979*: 21-23.

Ahlstrom, E. H. 1930. The shell bearing Mollusca of Michigan. *Nautilus* 6: 42-47.

Aldridge, D.W., B.S. Payne and A.C. Miller. 1987. The effects of intermittent exposure to suspended solids and turbulence on three species of freshwater mussels. *Environmental Pollution* 45:17-28.

Allen, W. R. 1914. The food and feeding habits of freshwater mussels. *Biological Bulletin* 27: 127-147.

Allen, W. R. 1921. Studies of the biology of freshwater mussels. *Biological Bulletin* 40: 210-241.

Anderson, K. B., Sparks, R. E. & A. A. Paparo. 1978. Rapid assessment of water quality using the fingernail clam, *Musculium transversum. University of Illinois, Water Resources Center, Urbana, UILU-WRC-78-0133, Research Report* (133). 115 pp.

Antipa, G. A. & E. B. Small. 1971. The occurrence of thigmotrichous ciliated protozoa inhabiting the mantle cavity of unionid molluscs of Illinois. *Transactions of the American Microscopical Society* 90: 463-472.

Arey, L. B. 1921. An experimental study on glochidia and the factors underlying encystment. *Journal of Experimental Zoölogy* 33: 463-499.

Arey, L. B. 1924a. Observations on an acquired immunity to a metazoan parasite. *Journal of Experimental Zoölogy* 38: 377-381.

Arey, L. B. 1924b. Glochidial cuticulae, teeth, and the mechanics of attachment. *Journal of Morphology and Physiology* 39: 323-335.

Arey, L. B. 1932a. The formation and structure of the glochidial cyst. *Biological Bulletin* 62: 212-221.

Arey, L. B. 1932b. The nutrition of glochidia during metamorphosis. *Journal of Morphology* 53: 201-221.

Arey, L. B. 1932c. A microscopical study of glochidial immunity. *Journal of Morphology* 53: 367-379.

Aughey, S. 1877. Catalogue of land and fresh-water shells of Nebraska. *Bulletin of the U. S. Geological and Geographical Survey* 3: 697-704.

Baker, F. C. 1918. The relation of shellfish to fish in Oneida Lake, New York. *New York State College of Forestry at Syracuse University, Circular* (21): 11-34, 16 figs.

Bangham, R. V. 1940. Parasites of fish of Algonquin Park lakes. *Transactions of the American Fisheries Society for 1940*: 161-171.

Bangham, R. V. 1955. Studies on fish parasites of Lake Huron and Manitoulin Island. *American Midland Naturalist* 53: 184-194.

Barney, R. L. 1922. An indication of the value of artificial propagation of pearly mussels. *Nautilus* 35: 53-58.

Bates, J. M. 1962. Impact of impoundment on the mussel fauna of Kentucky Reservoir, Tennessee River. *American Midland Naturalist* 68:232-236.

Bauer, G. 1986. The status of the freshwater pearl mussel *Margaritifera margaritifera* L. in the south of its European range. *Biological Conservation* 38: 1-9.

Bauer, G. 1987a. The parasitic stage of the freshwater pearl mussel (*Margaritifera margaritifera* L.). II. Susceptibility of brown trout. *Archiv für Hydrobiologie, Supplement* 76: 403-412.

Bauer, G. 1987b. The parasitic stage of the freshwater pearl mussel (*Margaritifera margaritifera* L.). III. Host relationships. *Archiv für Hydrobiologie, Supplement* 76: 413-423.

Bauer, G. 1987c. Reproductive strategy of the freshwater pearl mussel *Margaritifera margaritifera*. *Journal of Animal Ecology* 56: 691-704.

Bauer, G. 1988. Threats to the freshwater pearl mussel *Margaritifera margaritifera* L. in central Europe. *Biological Conservation* 45: 239-253.

Bauer, G. & C. Vogel. 1987. The parasitic stage of the freshwater pearl mussel (*Margaritifera margaritifera* L.). I. Host response to glochidiosis. *Archiv für Hydrobiologie, Supplement* 76: 393-402.

Baxter, R. M. 1977. Environmental effects of dams and impoundments. *Annual Review of Ecology and Systematics* 8:255-283.

Berrow, S. D. 1991. Predation by the hooded crow *Corvus corone cornix* on freshwater pearl mussels *Margaritifera margaritifera*. *Irish Naturalists' Journal* 23:492-493.

Bisbee, G. D. 1984. Ingestion of phytoplankton by two species of freshwater mussels, the black sandshell, *Ligumia recta*, and the three ridger, *Amblema plicata*, from the Wisconsin River in Oneida County, Wisconsin. *Bios* 55: 219-225.

Blystad, C. N. 1924. Significance of larval mantle of fresh-water mussels during parasitism, with notes on a new mantle condition exhibited by *Lampsilis luteola*. *Bulletin of the United States Bureau of Fisheries* **39**: 203-219.

Buchanan, A. C. 1987. Aspects of the life history of the Curtis' pearly mussel, *Epioblasma florentina curtisi* (Utterback 1915). *Final Report, Endangered Species Project SE-3-2, Missouri Department of Conservation.* 21 pp.

Buddensiek, V., Engel, H., Fleischauer-Rössing, S., & K. Wächtler. 1993. Studies on the chemistry of interstitial water taken from defined horizons in the fine sediments of bivalve habitats in several northern German lowland waters. II: Microhabitats of *Margaritifera margaritifera* L., *Unio crassus* (Philipsson) and *Unio tumidus* Philipsson. *Archiv für Hydrobiologie* 127: 151-166.

Chamberlain, T. K. 1934. The glochidial conglutinates of the Arkansas Fanshell, *Cyprogenia aberti* (Conrad). *Biological Bulletin* 66: 55-61.

Churchill, E. P. & S. I. Lewis. 1924. Food and feeding in fresh-water mussels. *Bulletin of the United States Bureau of Fisheries [Document 963]* **39**: 439-471.

Cicerello, R. R., Warren, M. L. & G. A. Schuster. 1991. A distributional checklist of the freshwater unionids (Bivalvia: Unionoidea) of Kentucky. *American Malacological Bulletin* **8**: 113-129.

Clark, H. W. & S. Stein. 1921. Glochidia in surface towings. *Nautilus* 35: 16-20.

Clark, H. W. & C. B. Wilson. 1912. The mussel fauna of the Maumee River. *U. S. Bureau of Commercial Fisheries, Bureau of Fisheries Document* (757): 1-72.

Clarke, A. H. 1967. Unionid introduction in Massachusetts: results. *Nautilus* **80**: 106-108.

Clarke, A. H. 1973. On the distribution of Unionidae in the Sydenham River, southern Ontario, Canada. *Malacological Review* **6**: 63-64.

Clarke, A. H. 1978. *The freshwater molluscs of Canada*. National Museums of Canada, Ottawa.

Clarke, A. H. 1986. The mesoconch: a record of juvenile life in Unionidae. *Malacology Data Net* **1**(2): 21-36.

Conrad, T. A. 1836. *Monography of the Family Unionidae, or Naiades of Lamarck, (Fresh Water Bivalve Shells,) of North America, Illustrated by Figures drawn on Stone from Nature*. J. Dobson, Philadelphia. pp. 17-64, pls. 6-35.

Convey, L. E., Hanson, J. M. & W. C. MacKay. 1989. Size-selective predation on unionid clams by muskrats. *Journal of Wildlife Management* **53**: 654-657.

Corwin, R. S. 1920. Raising freshwater mussels in enclosures. *Transactions of the American Fisheries Society* **49**: 81-84.

Corwin, R. S. 1921. Further notes on raising freshwater mussels in enclosures. *Transactions of the American Fisheries Society* **50**: 307-311.

Cummings, K. S. & J. M. K. Berlocher. 1990. The naiades or freshwater mussels (Bivalvia: Unionidae) of the Tippecanoe River, Indiana. *Malacological Review* **23**: 83-98.

Cummings, K. S., Meyer, C. A. & L. M. Page. 1992. *Survey of the Freshwater Mussels (Mollusca: Unionidae) of the Wabash River Drainage. Final Report.* Illinois Natural History Survey, Center for Biodiversity, Technical Report 1992 (1). 201 pp.

Dartnall, H. J. G. & M. Walkey. 1979. The distribution of glochidia of the swan mussel, *Anodonta cygnea* (Mollusca) on the three-spined stickleback *Gasterosteus aculeatus* (Pisces). *Journal of Zoology, London* **189**: 31-37.

Davids, C. 1973. The relations between mites of the genus *Unionicola* and the mussels *Anodonta* and *Unio*. *Hydrobiologia* **41**: 37-44.

Dean, G. W. 1890. Distribution of Unionidæ in the three rivers, Mahoning, Cuyahoga and Tuscarawas. *Nautilus* **4**: 20-22.

Downing, J. A. & W. L. Downing. 1992. Spatial aggregation, precision, and power in surveys of freshwater mussel populations. *Canadian Journal of Fisheries and Aquatic Sciences* **49**: 985-991.

Downing, J. A., Rochon, Y. & M. Pérusse. 1993. Spatial aggregation, body size, and reproductive success in the freshwater mussel *Elliptio complanata*. *Journal of the American Benthological Society* **12**: 148-156.

Ecological Specialists, Inc. 1992. *Final Report for the Tippecanoe River Unionid, Fish and Habitat Study.* Indiana Department of Natural Resources & U.S. Fish and Wildlife Service. 108 pp. + appendices.

Ecological Specialists, Inc. 1993a. *Mussel Habitat Suitability and Impact Analysis of the Tippecanoe River.* Indiana Department of Natural Resources & U.S. Fish and Wildlife Service. 102 pp.+ appendices.

Ecological Specialists, Inc. 1993b. *Final Report on a Unionid Investigation of a Proposed Pipeline Crossing on the Elk River, West Virginia.* Prepared for GAI Consultants, Inc., Monroeville, PA. 9 pp.

Ecological Specialists, Inc. 1993c. *Unionid Survey of the Lower Muskingum River.* Division of Wildlife, Ohio Department of Natural Resources, Columbus, OH. 51 pp. + appendices.

Ellis, M. M. 1936. Erosion silt as a factor in aquatic environments. *Ecology* **17**: 29-42.

Ellis, M. M. 1942. Fresh-water impoundments. *Transactions of the American Fisheries Society for 1941*: 80-93.

Evermann, B. W. & H. W. Clark. 1920. Lake Maxinkuckee. A physical and biological survey. *Indiana Department of Conservation Publication* (7): [Vol. 1] 660 pp, [Vol. 2] 512 pp.

Faussek, V. 1895. Ueber den Parasitismus der *Anodonta*-Larven in der Fischhaut. *Biologisches Centralblatt* **15**: 115-125.

Fikes, M. H. 1972. Maintenance of the naiad *Amblema plicata* (Say, 1817) in an artificial system. *Bulletin of the American Malacological Union for 1972*: 35.

Fischerstrom, I. 1761. De concharum margaritiferarum natura. *Commentarii de Rebus in Scientia Naturali et Medicina Gestis* **10**: 204, 205.

Fuller, S. L. H. 1971. A brief field guide to the fresh-water mussels (Mollusca: Bivalvia: Unionacea) of the Savannah River system. *Association of Southeastern Biologists Bulletin* **18**: 137-146.

Fustish, C. A. & R. E. Millemann. 1978. Glochidiosis of salmonid fishes. II. Comparison of tissue response of Coho and Chinook Salmon to experimental infection with *Margaritifera margaritifera* (L.) (Pelecypoda: Margaritanidae). *Journal of Parasitology* **64**: 155-157.

Gardiner, D. B., Silverman, H. & T. H. Dietz. 1991. Musculature associated with the water canals in freshwater mussels and response to monamines *in vitro*. *Biological Bulletin* **180**: 453-465.

Gatenby, C. M., Neves, R. J. & B. C. Parker. 1993. Preliminary observations from a study to culture recently metamorphosed mussels. *Bulletin of the North American Benthological Society* **10**: 128 [abstract].

Goodrich, C. 1914. Union of the Wabash and Maumee drainage systems. *Nautilus* **27**: 131, 132.

Gordon, M. E. & J. B. Layzer. 1989. Mussels (Bivalvia: Unionoidea) of the Cumberland River. Review of life histories and ecological relationships. *United States Fish and Wildlife Service, Biology Report* **89**(15). 99 pp.

Gordon, M. E. & D. G. Smith. 1990. Autumnal reproduction in *Cumberlandia monodonta* (Unionoidea: Margaritifera). *Transactions of the American Microscopical Society* **109**: 407-411.

Gordon, M. J., Swan, B. K. & C. G. Patterson. 1978. *Baeoctenus bicolor* (Diptera: Chironomidae) parasitic in unionid bivalve molluscs, and notes on other chironomid-bivalve associations. *Journal of the Fisheries Research Board of Canada* **35**: 154-157.

Goudraeu, S. E., Neves, R. J. & R. J. Sheehan. 1993. Effects of wastewater treatment plant effluents on freshwater mollusks in the upper Clinch River, Virginia, USA. *Hydrobiologia* **252**: 211-230.

Grier, N. M. & J. F. Mueller. 1922. Notes on the naiad fauna of the upper Mississippi River. II. The naiades of the upper Mississippi drainage. *Nautilus* 35: 46-49, 96-103.

Haag, K. H. & J. H. Thorp. 1991. Cross-channel distribution patterns of zoobenthos in a regulated reach of the Tennessee River. *Regulated Rivers: Research & Management* **6**: 225-233.

Hanson, J. M., MacKay, W. C. & E. E. Prepas. 1988. The effects of water depth and density on the growth of a unionid clam. *Freshwater Biology* 19: 345-355.

Hanson, J. M., MacKay, W. C. & E. E. Prepas. 1989. Effect of size-selective predation by muskrats (*Ondatra zebithicus*) on a population of unionid clams (*Anodonta grandis simpsonianus*). *Journal of Animal Ecology* 58: 15-28.

Harman, W.N. 1974. The effects of reservoir construction and channelization on the mollusks of the upper Delaware watershed. *Bulletin of the American Malacological Union for 1974*:12-14.

Havlik, M. E. & L. L. Marking. 1987. Effects of contaminants on naiad mollusks (Unionidae): a review. *U. S. Department of the Interior, Fish and Wildlife Service Resource Publication* (164). 20 pp.

Heard, W. H. 1979. Hermaphroditism in *Elliptio* (Pelecypoda: Unionidae). *Malacological Review* **12**: 21-28.

Hendrix, S. S., Vidrine, M. F. & R. H. Hartenstine. 1985. A list of records of freshwater aspidogastrids (Trematoda) and their hosts in North America. *Proceedings of the Helminthological Society of Washington* **52**: 289-296.

Hoggarth, M. A. 1987. The freshwater mussels (Unionidae) of the upper St. Joseph River basin within Ohio. *Final Report to the Division of Wildlife, Ohio Department of Natural Resources*. 73 pp.

Hove, M. & R. Neves. 1991. Distribution and life history of the James River spinymussel. *Endangered Species Technical Bulletin* **16**: 9.

Howard, A. D. 1914a. Experiments in propagation of fresh-water mussels of the *Quadrula* group. *Report of the Commissioner of Fisheries for the Fiscal Year 1913, Appendix 4 [Document 801]*: 1-52; 6 pls.

Howard, A. D. 1914b. A second case of metamorphosis without parasitism in the Unionidae. *Science* **40**: 353-355.

Howard, A. D. 1914c. A new record in rearing fresh-water mussels. *Transactions of the American Fisheries Society* **44**: 45-47.

Howard, A. D. 1915. Some exceptional cases of breeding among the Unionidae. *Nautilus* **29**: 4-11.

Howard, A. D. 1916. A second generation of artificially reared fresh-water mussels. *Transactions of the American Fisheries Society* 46: 89-92.

Howard, A. D. 1922. Experiments in the culture of fresh-water mussels. *Bulletin of the United States Bureau of Fisheries [Document 916]* **38**: 63-89.

Howard, A. D. & B. J. Anson. 1923. Phases in the parasitism of the Unionidae. *Journal of Parasitology* 9: 68-82; pls. 7, 8.

Hudson, R. G. & B. G. Isom. 1984. Rearing juveniles of the freshwater mussels (Unionidae) in a laboratory setting. *Nautilus* **98**: 129-135.

Huebner, J. D. & K. S. Pynnönen. 1992. Viability of glochidia of *Anodonta* exposed to low pH and selected metals. *Canadian Journal of Zoology* **70**: 2348-2355.

Huehner, M. K. & C. L. Corr. 1994. The unionid mussel fauna of Pymatuning Creek in Ashtabula County, Ohio. *Final Report to the Ohio Division of Natural Areas and Preserves.* 14 pp.

Humphrey, C. 1987a. Effects of mine waters on freshwater mussels. Pp. 100-103. *[In:] Alligator Rivers Region Research Institute, Annual Research Summary for 1985-86.* Australian Government Publishing Service, Canberra. 142 pp.

Humphrey, C. 1987b. Freshwater Mussels (*Velesunio angasi*). Pp. 107, 108. *[In:] Alligator Rivers Region Research Institute, Annual Research Summary for 1986-1987.* Australian Government Publishing Service, Canberra.

Humphrey, C. 1988. Development of creekside and *in situ* monitoring systems. Pp. 80-? *[In:] Alligator Rivers Region Research Institute, Annual Research Summary for 1987-1988.* Australian Government Publishing Service, Canberra.

Humphrey, C. L. & R. D. Simpson. 1985. The biology and ecology of *Velesunio angasi* (Bivalvia: Hyriidae) in the Malaga Creek, Northern Territory. *Report to the Office of Supervising Scientist for the Alligator Rivers Region, Open File Record* (38): 476 pp.; 2 appendices.

Imlay, M. J. 1972. Greater adaptability of freshwater mussels to natural rather than to artificial displacement. *Nautilus* **86**: 76-79.

Imlay, M. J. & M. L. Paige. 1972. Laboratory growth of freshwater sponges, unionid mussels, and sphaeriid clams. *Progressive Fish-Culturist* **34**: 210-216.

Isely, F. B. 1914. Experimental study of the growth and migration of fresh-water mussels. *Report of the Commissioner of Fisheries for the Fiscal Year 1913, Appendix 3 [Document 792]*: 1-24; 3 pls.

Isom, B. G. 1983. Potential uses of *in vitro* culture of freshwater mussel glochidia for conservation. Pp. 42, 43. *[In:]* Miller, A. C. (ed.), *Report of Freshwater Mussels Workshop*, U.S. Army Engineer Waterways Experiment Station, Environmental Laboratory, Vicksburg, Mississippi.

Isom, B. G. 1986a. *Activity 4: Artificial culture.* Tennessee Valley Authority Cumberlandian Mollusk Conservation Program, Norris, Tennessee.

Isom, B. G. 1986b. Systems culture of freshwater shellfish (bivalves). *EIFAC/FAO Symposium on Selection, Hybridization and Genetic Engineering in Aquaculture of Fish and Shellfish for Consumption and Stocking, Bordeaux, France, 27-30 May 1986.* 28 pp.

Isom, B. G. & C. Gooch. 1986. Rationale and sampling designs for freshwater mussels Unionidae in streams, large rivers, impoundments, and lakes. Pp. 46-59. *[In:]* B. G. Isom (ed.), Rationale for sampling and interpretation of ecological data in the assessment of freshwater ecosystems. *American Society for Testing and Materials, Special Technical Paper* (894).

Isom, B. G. & R. G. Hudson. 1982. *In vitro* culture of parasitic mussel glochidia. *Nautilus* **96**: 147-151.

Isom, B. G. & R. G. Hudson. 1984. Freshwater mussels and their fish hosts; physiological aspects. *Journal of Parasitology* **70**: 318, 319.

Ison [*sic*], B. G. & R. G. Hudson. 1984. Culture of freshwater mussel glochidia in an artificial habitat utilizing complex liquid growth media. *United States Patent 4,449,480*: 18 pp.

Jacobson, P. J., Farris, J. L., Cherry, D. S. & R. J. Neves. 1993. Juvenile freshwater mussel (Bivalvia: Unionidae) responses to acute toxicity testing with copper. *Environmental Toxicology and Chemistry* **12**: 879-883.

Jansen, W. A. & J. M. Hanson. 1991. Estimates of the number of glochidia produced by clams (*Anodonta grandis simpsonianus* Lea), attaching to yellow perch (*Perca flavescens*), and surviving to various ages in Narrow Lake, Alberta. *Canadian Journal of Zoology* **69**: 973-977.

Jenkinson, J. J. & J. H. Heuer. 1986. *Activity 9: Selection of transplant sites and habitat characterization*. Tennessee Valley Authority Cumberlandian Mollusk Conservation Program, Norris, Tennessee.

Jeong, K.-H., Min, B.-J. & P.-R. Chung. 1993. An anatomical study of the glochidium of *Anodonta arcaeformis*. *Malacological Review* **26**: 71-79.

Johnson, R. I. 1978. Systematics and zoogeography of *Plagiola (= Dysnomia = Epioblasma)*, an almost extinct genus of freshwater mussels (Bivalvia: Unionidae) from middle North America. *Bulletin of the Museum of Comparative Zoology* **148**: 239-321.

Johnson, I. C., Keller, A. E. & S. G. Zam. 1993. A method for conducting acute toxicity tests with the early life stages of freshwater mussels. Pp. 381-396. *[in:]* Landis, W. G., Hughes, J. S. & M. A. Lewis (eds.), *Environmental Toxicology and Risk Assessment; American Society for Testing and Materials, Standard Technical Publication* (1179).

Kat, P. W. 1983. Sexual selection and simultaneous hermaphroditism among the Unionidae (Bivalvia: Mollusca). *Journal of Zoology, London* **201**: 395-416.

Kays, W. T., Silverman, H. & T. H. Dietz. 1990. Water channels and water canals in the gill of the freshwater mussel, *Ligumia subrostrata*: ultrastructure and histochemistry. *Journal of Experimental Zoology* **254**: 256-269.

Keller, A. E. 1993. Acute toxicity of several pesticides, organic compounds and a wastewater effluent to the freshwater mussel, *Anodonta imbecilis, Ceriodaphnia dubia* and *Pimephales promelas. Bulletin of Environmental Contamination and Toxicology* **51**: 696-702.

Keller, A. E. & S. G. Zam. 1990. Simplification of *in vitro* culture techniques for freshwater mussels. *Environmental Toxicology and Chemistry* **9**: 1291-1296.

Keller, A. E. & S. G. Zam. 1991. The acute toxicity of selected metals to the freshwater mussel, *Anodonta imbecillis. Environmental Toxicology and Chemistry* **10**: 539-546.

Koch, L. M. 1990. Reintroduction of *Potamilus capax* to portions of the upper Mississippi River in Missouri. *Proceedings of the 46th Annual Meeting of the Upper Mississippi River Conservation Committee*: 88-99.

Kovalak, W. P., Dennis, S. D. & J. M. Bates. 1986. Sampling effort required to find rare species of freshwater mussels. Pp. 34-45. *[In:]* B. G. Isom (ed.), Rationale for sampling and interpretation of ecological data in the assessment of freshwater ecosystems. *American Society for Testing and Materials, Special Technical Paper* (894).

Kwon, O.-K., Park, G.-M., Lee, J.-S. & H.-B. Song. 1993. Scanning electron microscope studies of the minute shell structure of glochidia of three species of Unionidae (Bivalvia) from Korea. *Malacological Review* **26**: 63-70.

La Rocque, A. 1953. Catalogue of the Recent Mollusca of Canada. *National Museum of Canada, Bulletin* (129): 1-406.

Lamarck, C. de. 1819. *Histoire naturelle des animaux sans vertèbres*. **6**. Paris. 643 pp.

Lea., I. 1837. Descriptions of new freshwater and land shells. *Transactions of the American Philosophical Society* 6: 95-102.

Lea, I. 1863. *Observations of the Genus* Unio, *together with Descriptions of New Species, their Soft Parts, and Embryonic Forms, in the Family Unionidae*, **10**. Privately printed, Philadelphia. 94 pp.

Lefevre, G. & W. C. Curtis. 1910. Experiments in the artificial propagation of fresh-water mussels. *Bulletin of the United States Bureau of Fisheries [Document 671]* **28**: 615-626.

Lefevre, G. & W. C. Curtis. 1911. Metamorphosis without parasitism in the Unionidæ. *Science* **33**: 863-865.

Lemon, J. H. 1898. Interglacial shells at Toronto, Canada. *Nautilus* **12**: 6-7.

Liquori, V. M. & G. D. Insler. 1985. Gill parasites of the white perch: phenologies in the lower Hudson River. *New York Fish and Game Journal* **32**: 71-76.

Lynn, J. W. 1987. Release of motile spermatophores from the freshwater mussel *Anodonta grandis*. *American Zoologist* **27**: 90A [abstract].

Mackie, G. L. & J. M. Topping. 1988. Historical changes in the unionid fauna of the Sydenham River watershed and downstream changes in shell morphometrics of three common species. *Canadian Field-Naturalist* **102**: 617-626.

Mäkelä, T. P. & A. O. J. Oikari. 1992. The effects of low water pH on the ionic balance in freshwater mussel *Anodonta anatina* L. *Annales Zoologici Fennici* **29**:169-175.

Marking, L. L. & T. D. Bills. 1980. Acute effects of silt and sand sedimentation on freshwater mussels. Pp. 204-211. *[In:]* Rasmussen, J. L. (ed.), *Proceedings of the symposium on upper Mississippi River bivalve mollusks*. Upper Mississippi River Conservation Committee, Rock Island, IL.

Meyers, T. R., Millemann, R. E. & C. A. Fustish. 1980. Glochidiosis of salmonid fishes. IV. Humoral and tissue responses of coho and chinook salmon to experimental infection with *Margaritifera margaritifera* (L.) (Pelecypoda: Margaritanidae). *Journal of Parasitology* **66**: 274-281.

Miller, A. C. & B. S. Payne. 1988. The need for quantitative sampling to characterize size demography and density of freshwater mussel communities. *American Malacological Bulletin* **6**: 49-54.

Miller, A. C. & B. S. Payne. 1993. Qualitative versus quantitative sampling to evaluate population and community characteristics at a large-river mussel bed. *American Midland Naturalist* **130**: 133-145.

Miller, A. C., Payne, B. S. & R. Tippit. 1992. Characterization of a freshwater mussel (Unionidae) community immediately downriver of Kentucky Lock and Dam in the Tennessee River. *Transactions of the Kentucky Academy of Science* **53**: 154-161.

Mitchell, R. & J. L. Wilson. 1965. New species of water mites (*Unionicola*) from Tennessee unionid mussels. *Journal of the Tennessee Academy of Sciences* **40**: 104-107.

Neel, J. K. 1963. Impact of reservoirs. Pp. 575-593. *[In:]* D.G. Frey (ed.), *Limnology in North America*, University of Wisconsin Press, Madison.

Negus, C. L. 1966. A quantitative study of growth and production of unionid mussels in the River Thames at Reading. *Journal of Animal Ecology* **35**: 513-532.

Nelson, D. 1982. Relocation of *Lampsilis higginsi* in the upper Mississippi River. Pp. 104-107. *[In:]* Miller, A. C. (ed.), *Report of Freshwater Mollusks Workshop*, U.S. Army Engineer Waterways Experiment Station, Environmental Laboratory, Vicksburg, Mississippi.

Neves, R. J. 1983. The status of freshwater mussel research in Virginia. Pp. 155-168. *[In:]* Miller, A. C. (ed.), *Report of Freshwater Mussels Workshop*, U.S. Army Engineer Waterways Experiment Station, Environmental Laboratory, Vicksburg, MS.

Neves, R. J. & Odum, M. C. 1989. Muskrat predation on endangered freshwater mussels in Virginia. *Journal of Wildlife Management* **53**: 934-941.

Neves, R. J. & J. C. Widlak. 1988. Occurrence of glochidia in stream drift and on fishes of the Upper North Fork Holston River, Virginia. *American Midland Naturalist* **119**: 111-120.

Ortmann, A. E. 1910. The discharge of the glochidia in the Unionidae. *Nautilus* **24**: 94, 95.

Ortmann, A. E. 1912. Notes upon the families and genera of the najades. *Annals of the Carnegie Museum* **8**: 222-365.

Ortmann, A. E. 1919. A monograph of the naiades of Pennsylvania. Part III. Systematic account of the genera and species. *Memoirs of the Carnegie Museum* **8**: 1-385.

Parker, R. S., Hackney, C. T. & M. F. Vidrine. 1979. Notes on the reproductive biology of *Glebula rotundata* (Lamarck) (Bivalvia: Unionidae: Lampsilinae). *Bulletin of the American Malacological Union for 1979*: 66, 67 [abstract].

Parker, R. S., Hackney, C. T. & M. F. Vidrine. 1984. Ecology and reproductive strategy of a south Louisiana freshwater mussel, *Glebula rotundata* (Lamarck) (Unionidae: Lampsilini). *Freshwater Invertebrate Biology* **3**: 53-58.

Parmalee, P. W. & M. H. Hughes. 1993. Freshwater mussels (Mollusca: Pelecypoda: Unionidae) of Tellico Lake: Twelve years after impoundment of the Little Tennessee River. *Annals of Carnegie Museum* **62**:81-93.

Poupart, M. 1706. Remarques sur les coquillages à deux coquilles, & premierement sur les moules. *Histoire de l'Academie Royale des Sciences, Paris, 1706*: 51-61.

Pynnönen, K. 1990. Effect of acidic conditions on cadmium kinetics and electrolyte balance in the freshwater clam *Unio pictorum*. *Annales Zoologici Fennici* **27**: 351-360.

Rand, T. G. & M. Wiles. 1982. Species differentiation of the glochidia of *Anodonta cataracta* Say, 1817 and *Anodonta implicata* Say, 1829 (Mollusca: Unionidae) by scanning electron microscopy. *Canadian Journal of Zoology* **60**: 1722-1727.

Rees, W. J. 1965. The aerial dispersal of Mollusca. *Proceedings of the Malacological Society of London* 36: 269-282.

Reuling, F. H. 1919. Acquired immunity to an animal parasite. *Journal of Infectious Diseases* **24**: 337-346.

Richard, P. E., Dietz, T. H. & H. Silverman. 1991. Structure of the gill during reproduction in the unionids *Anodonta grandis, Ligumia subrostrata*, and *Carunculina parva texasensis*. *Canadian Journal of Zoology* **69**: 1744-1754.

Ridley, J. E. & J. A. Steel. 1975. Ecological aspects of river impoundments. *[In:]* Whitton, B. A., (ed.), *River Ecology, Studies in Ecology* **2**:565-587.

Salmon, A. & R. H. Green. 1983. Environmental determinants of unionid clam distribution in the Middle Thames River, Ontario. *Canadian Journal of Zoology* **61**:832-838.

Schierholz, C. 1889. Über Entwicklung der Unioniden. *Denkschriften der Kaiserlichen Akademie der Wissenschaften, Wien. Mathematisch-Naturwissenschafliche Classe* 55: 183-214; 4 pls.

Sherman, R. A. 1993. Glochidial release and reproduction of the snuffbox mussel, *Epioblasma triquetra*; timing in southern Michigan. *Bulletin of the North American Benthological Society* **10**: 197 [abstract].

Silverman, H., Steffens, W. L. & T. H. Dietz. 1985. Calcium from extracellular concretions in the gills of freshwater unionid mussels is mobilized during reproduction. *Journal of Experimental Zoology* **236**: 137-147.

Simmons, G. M. & J. R. Reed. 1973. Mussels as indicators of biological recovery zone. *Journal of the Water Pollution Control Federation* **45**: 2480-2492.

Simpson, C. T. 1900. *A descriptive catalogue of the naiades or pearly fresh-water mussels*. B. Walker, Detroit, Michigan. 1539 pp.

Smith, D. G. 1979. Marsupial anatomy of the demibranch of *Margaritifera margaritifera* (Lin.) in northeastern North America (Pelecypoda: Unionacea). *Journal of Molluscan Studies* **45**: 39-44.

Stansbery, D. H. 1961. The naiades (Mollusca, Pelecypoda, Unionacea) of Fishery Bay, South Bass Island, Lake Erie. *Sterkiana* (5): 1-37.

Stansbery, D. H. 1973. Dams and the extinction of aquatic life. *Garden Club of America Bulletin* **61**:43-46.

Stansbery, D. H. & C. C. King. 1983. Management of Muskingum River mussel (unionid mollusk) populations. *Ohio State University Museum of Zoology Reports for 1983* (5). 79 pp.

Strayer, D. 1979. Some recent collections of mussels from southeastern Michigan. *Malacological Review* **12**: 93-95.

Surber, T. 1913. Notes on the natural hosts of fresh-water mussels. *Bulletin of the United States Bureau of Fisheries [Document 778]* **32**: 101-115; pls. 29-31.

Tankersley, R. A. & R. V. Dimock. 1992. Quantitative analysis of the structure of the marsupial gills of the freshwater mussel *Anodonta cataracta. Biological Bulletin* **182**: 145-154.

Turgeon, D. D., Bogan, A. E., Coan, E. V., Emerson, W. K., Lyons, W. G., Pratt, W. L., Roper, C. F. E., Scheltema, A., Thompson, F. G. & J. D. Williams. 1988. Common and scientific names of aquatic invertebrates from the United States and Canada: Mollusks. *American Fisheries Society Special Publication* (16). 277 pp.

Utterback, W. I. 1931. Sex behavior among naiades. *Proceedings of the West Virginia Academy of Science* **5**: 43-45.

van der Schalie, H. 1966. Hermaphroditism among North American freshwater mussels. *Malacologia* 5: 77, 78.

van der Schalie, H. 1970. Hermaphroditism among North American freshwater mussels. *Malacologia* **10**: 93-112.

Walker, B. 1913. The unione fauna of the Great Lakes. *Nautilus* **27**: 18-23.

Waller, D. L. & L. G. Mitchell. 1989. Gill tissue reactions in walleye *Stizostedion vitreum vitreum* and common carp *Cyprinus carpio* to glochidia of the freshwater mussel *Lampsilis radiata siliquoidea*. *Diseases of Aquatic Organisms* 6: 81-87.

Watters, G. T. 1986. A survey of the unionid molluscs of the Big Darby Creek System in Ohio. *Final Report to The Nature Conservancy*. 149 pp.

Watters, G. T. 1988a. The naiad fauna of selected streams in Ohio. I. Stillwater River of Miami River. II. Stream systems of south central Ohio from the Little Miami River to the Hocking River, excluding the Scioto River proper. *Final Report to the Division of Wildlife, Ohio Department of Natural Resources*. 440 pp.

Watters, G. T. 1988b. A survey of the freshwater mussels of the St. Joseph River system, with emphasis on the federally endangered White Cat's Paw Pearly Mussel. *Final Report to the Division of Fish and Wildlife, Indiana Department of Natural Resources*. 127 pp.

Watters, G. T. 1990. 1990 Survey of the unionids of the Big Darby Creek system. *Final Report to The Nature Conservancy*. 229 pp.

Watters, G. T. (in press). The unionid fauna of the Big Darby Creek system in Ohio. *Malacological Review*.

Weaver, L. R., Pardue, G. R. & R. J. Neves. 1991. Reproductive biology and fish hosts of the Tennessee clubshell *Pleurobema oviforme* (Mollusca: Unionidae) in Virginia. *American Midland Naturalist* **126**: 82-89.

Weir, G. P. 1977. An ecology of the Unionidae in Otsego Lake with special references to immature stages. *State University College at Oneonta, Biological Field Station, Occasional Paper* (4): 1-108.

Williams, J. D., Fuller, S. L. H. & R. Grace. 1992. Effects of impoundments on freshwater mussels (Mollusca: Bivalvia: Unionidae) in the main channel of the Black Warrior and Tombigbee Rivers in western Alabama. *Bulletin of the Alabama Museum of Natural History* (13): 1-10.

Winslow, M. L. 1918. *Pleurobema clava* (Lam.) and *Planorbis dilatatus buchanensis* (Lea) in Michigan. *Occassional Papers of the Museum of Zoology, University of Michigan* (51): 1-4.

Yeager, B. L. 1986. *Epioblasma brevidens, Epioblasma capsaeformis* and *Epioblasma triquetra*. Pp. 15, 16, 40-49. *[In]:* Hill, D. M. (ed.), *Activity 3: Identification of fish hosts*. Tennessee Valley Authority Cumberlandian Mollusk Conservation Program, Norris, TN.

Yeager, M. M., Cherry, D. S. & R. Neves. 1993. Interstitial feeding behavior of juvenile unionid mussels. *Association of Southeastern Biologists Bulletin* 40: 113 [abstract].

Young, D. 1911. The implantation of the glochidium on the fish. *University of Missouri Bulletin, Science Series* 2: 1-16, 3 pls.

Young, M. & J. Williams. 1984. The reproductive biology of the freshwater pearl mussel *Margaritifera margaritifera* (Linn.) in Scotland. I. Field studies. *Archiv für Hydrobiologie* 99: 405-422.

Young, M., Purser, G. J. & B. al-Mousawi. 1987. Infection and successful reinfection of brown trout [*Salmo trutta* (L.)] with glochidia of *Margaritifera margaritifera* (L.). *American Malacological Bulletin* 5: 125-128.

Zale, A. V. & R. J. Neves. 1982. Reproductive biology of four freshwater mussel species (Mollusca: Unionidae) in Virginia. *Freshwater Invertebrate Biology* 1: 17-28.

PART III: IMPLEMENTATION

The Implementation Schedule lists and ranks tasks that should be undertaken within the next three years in order to implement recovery of *Pleurobema clava* and *Epioblasma torulosa rangiana*. This schedule will be reviewed annually until the recovery objective is met, and priorities and tasks will be subject to revision. Tasks are presented in order of priority.

<u>Key to Implementation Schedule Column 1</u>

Task priorities are set according to the following standards:

Priority 1: Those actions that must be taken to prevent extinction or to prevent the species from declining irreversibly in the foreseeable future.

Priority 2: Those actions that must be taken to prevent a significant decline in species population, or some other significant impact short of extinction.

Priority 3: All other actions necessary to provide for full recovery of the species.

<u>Key to Agency Designations in Column 5</u>

USFWS	=	U.S. Fish and Wildlife Service
R3, R4, R5	=	Regions 3, 4, and 5, U.S. Fish and Wildlife Service
ES	=	Division of Ecological Services, U.S. Fish and Wildlife Service
FI	=	Division of Fisheries, U.S. Fish and Wildlife Service
EC	=	Division of Environmental Contaminants, U.S. Fish and Wildlife Service
EPA	=	Environmental Protection Agency
COE	=	U.S. Army Corps of Engineers
NPS	=	National Park Service
FS	=	U.S. Forest Service
NBS	=	National Biological Service
SRA	=	State natural resource agencies
TNC	=	The Nature Conservancy
CO	=	Other conservation organizations and land trusts
AI	=	Academic institutions
PO	=	Private organizations, including research institutes

IMPLEMENTATION SCHEDULE
Clubshell and Northern Riffleshell Recovery Plan

September 1994

Priority	Task Description	Task Number	Duration	Responsible Agency		Cost Estimates (000)			Comments
				USFWS	Other	FY1	FY2	FY3	
1	Delineate the ecosystems for the priority drainages identified for each species.	1.1	5 years	R3/R4/R5 ES, EC, FI	NBS SRA TNC	10	10	10	+ 10K/yr for FY4 and FY5.
1	As a near-term conservation measure, identify and participate in ongoing environmental planning and regulatory compliance processes within each ecosystem.	1.4	ongoing	R3/R4/R5 ES	EPA COE SRA	9	6		+ 30K total over FY4-FY25.
1	Monitor population status, including demographics, at existing sites through a collecting protocol.	2.1	ongoing	R3/R4/R5 ES	SRA TNC	50	50	50	+ 15K/yr for FY4-FY25.
1	Identify and map both actual and potential threats at existing sites and potential translocation sites.	2.2	ongoing	R3/R4/R5 ES	EPA NBS SRA	25	25	25	Spot sampling over recovery period in conjunction with Task 2.1. 125K total projected cost for FY4-FY25.
1	Enforce all laws and regulations pertaining to the collection of specimens and protection of mussel habitat.	2.3	ongoing	R3/R4/R5 ES	EPA COE SRA				No itemized costs.
1	Determine contaminant sensitivity for each life stage.	3.1	4 years	R3/R4/R5 ES	EPA	30	30	30	+ 30K in FY4.
1	Complete life history studies for each species.	3.2	2 years	R3/R4/R5 ES	AI		50	50	Possible follow-up studies as needed. Costs not included.
1	Identify the potential effects of, and responses to, zebra and/or quagga mussel invasions, and their control measures.	3.5	periodic	R3/R4/R5 ES, FI	SRA		10		+ 10K/yr for 3 years. This task will rely in large part on studies conducted under other auspices.

Clubshell and Northern Riffleshell Recovery Plan Implementation Schedule (continued), September 1994

Priority	Task Description	Task Number	Duration	Responsible Agency		Cost Estimates (000)			Comments
				USFWS	Other	FY1	FY2	FY3	
3	Identify other resource values within each ecosystem.	1.2	2 years	R3/R4/R5 ES, FI	SRA TNC CO AI	9		6	
3	Undertake genetic studies to determine the species' limits.	3.4	2 years		AI PO		12	12	
3	Release additional hosts.	4.3	3 years	R3/R4/R5 ES, FI	SRA				Not likely to be initiated within the first 3 fiscal years. Total projected cost of 50K.
3	Monitor new populations.	4.4	20 years	R3/R4/R5 ES	SRA			15	+ 15K/yr for FY4-FY22.
3	Distribute an educational video.	5.1	ongoing	R3/R4/R5 ES	NPS FS SRA				A freshwater mussel video has been developed.
3	Develop and distribute educational materials.	5.4	ongoing	R3/R4/R5 ES	NPS FS SRA		8		+ 8K/yr over 10 years, periodically.
3	Use media opportunities to reach the general public.	5.5	ongoing	R3/R4/R5 ES	SRA				No itemized costs.
3	Make presentations.	5.6	ongoing	R3/R4/R5 ES	SRA	2	1	1	+ 1K/yr for FY4-FY25.

Clubshell and Northern Riffleshell Recovery Plan Implementation Schedule (continued), September 1994

Priority	Task Description	Task Number	Duration	Responsible Agency USFWS	Responsible Agency Other	Cost Estimates (000) FY1	FY2	FY3	Comments
2	Identify activities or practices within each ecosystem that may affect the clubshell and northern riffleshell as well as other sensitive resources.	1.3	2 years	R3/R4/R5 ES, EC, FI	EPA SRA	9	6		
2	Help develop and implement comprehensive watershed plans.	1.5	8 years	R3/R4/R5 ES	SRA	50	50	50	+ 50K/yr for 5 more years (planning for approximately 2 drainages/yr).
2	Conduct site-specific protection and management programs.	2.4	ongoing	R3/R4/R5 ES, FI	EPA NPS FS SRA TNC	10	10	10	+ 10K/yr for FY4-FY25.
2	Conduct searches, as warranted, for additional populations.	2.5	3 years	R3/R4/R5 ES, EC, FI	SRA TNC	25	25	25	
2	Characterize the habitat that best supports these species.	3.3	2 years	R3/R4/R5 ES	NBS SRA AI PO		20	20	Acquire data in conjunction with Tasks 2.1, 2.2, 3.1, and 3.2.
2	Select potential translocation sites.	4.1	2 years	R3/R4/R5 ES	SRA TNC CO AI			10	+ 10K in FY4.
2	Translocate portions of existing populations.	4.2	10 years	R3/R4/R5 ES, FI	SRA			35	+ 35K/yr for 9 years.
2	Increase public and agency awareness of existing laws that protect these species.	5.2	3 years	R3/R4/R5 ES	SRA	3	3	3	
2	Encourage public involvement in the recovery process.	5.3	5 years	R3/R4/R5 ES	SRA TNC	5	5	5	+ 5K/yr for FY4 and FY5.

APPENDIX: LIST OF REVIEWERS

The following individuals and agencies submitted comments on the Technical/Agency draft of the Clubshell and Northern Riffleshell Recovery Plan. All letters of comment and responses are on file in the U.S. Fish and Wildlife Service's West Virginia Field Office, Elkins, West Virginia.

Robert M. Anderson
Aquatic Nongame Biologist
Indiana Department of Natural Resources
Division of Fish and Wildlife
402 West Washington Street
Indianapolis, Indiana 46204

Charles W. Bier
Natural Science and Stewardship Department
Western Pennsylvania Conservancy
Pittsburgh, Pennsylvania 15222

Richard G. Biggins
U.S. Fish and Wildlife Service
Ashville Field Office
330 Ridgefield Court
Asheville, North Carolina 28806

Ronald R. Cicerello
Kentucky State Nature Preserves Commission
407 Broadway
Frankfort, Kentucky 40601

Heidi L. Dunn
Ecological Specialists, Inc.
95 Algana Court
St. Peters, Missouri 63376

Raymond L. Hasse
Water Pollution Biologist
Water Management
Pennsylvania Department of Environmental Resources
Northwest Regional Office
1012 Water Street
Meadville, Pennsylvania 16335

Michael O. Holm
National Park Service
Acting Superintendent
Mammoth Cave National Park
Mammoth Cave, Kentucky 42259

David C. Hudak, Supervisor
Jennifer Szymanski, Endangered Species Biologist
U.S. Fish and Wildlife Service
Bloomington Field Office
620 South Walker Street
Bloomington, Indiana 47403

Kevin R. Kelly
Water Pollution Biologist
Pennsylvania Department of Environmental Resources
Bureau of Water Quality Management
Post Office Box 8465
Harrisburg, Pennsylvania 17105

Laura M. Knoth
Natural and Environmental Resources
Kentucky Farm Bureau Federation
9201 Bunsen Parkway
Post Office Box 20700
Louisville, Kentucky 40250

Kent E. Kroonemeyer, Supervisor
Buddy B. Fazio, Endangered Species Biologist
U.S. Fish and Wildlife Service
Reynoldsburg Ohio Field Office
6950-H American Parkway
Reynoldsburg, Ohio 43068

Robert McCance, Jr.
Director
Kentucky State Nature Preserves Commission
407 Broadway
Frankfort, Kentucky 40601

Mary Rabe
Michigan Natural Features Inventory
Stevens T. Mason Building
Post Office Box 30028
Lansing, Michigan 48909

T. Rooney
M. Moran
D. Weigman
T. Callaghan
Ecology Program, Department of Biology
University of Delaware
Newark, Delaware 19716

Thomas Rooney
Staff Ecologist
Preserve Appalachian Wilderness
57 Choate Street
Newark, Delaware 19711

Robin Smith
Executive Director
In Defense of Endangered Species
Post Office Box 21314
Columbus, Ohio 43221

For further information regarding this recovery plan, please contact:

Bill Tolin
U.S. Fish and Wildlife Service
West Virginia Field Office
Route 250 South, Elkins Shopping Plaza
Elkins, West Virginia 26241
telephone (304) 636-6586