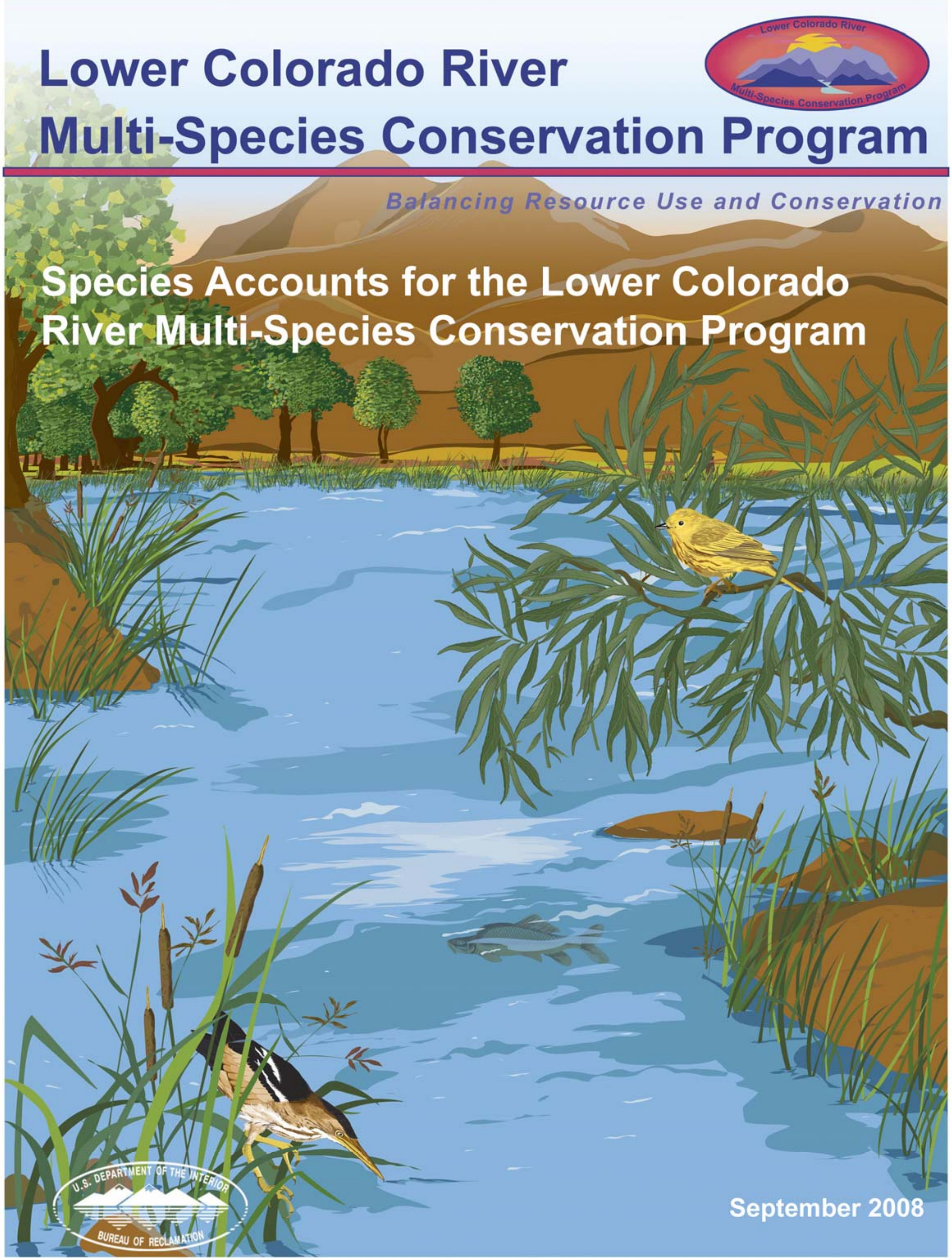


Lower Colorado River Multi-Species Conservation Program

Balancing Resource Use and Conservation

Species Accounts for the Lower Colorado River Multi-Species Conservation Program

September 2008

U.S. Department of the Interior
Bureau of Reclamation

Lower Colorado River Multi-Species Conservation Program Steering Committee Members

Federal Participant Group

Bureau of Reclamation
U.S. Fish and Wildlife Service
National Park Service
Bureau of Land Management
Bureau of Indian Affairs
Western Area Power Administration

Arizona Participant Group

Arizona Department of Water Resources
Arizona Electric Power Cooperative, Inc.
Arizona Game and Fish Department
Arizona Power Authority
Central Arizona Water Conservation District
Cibola Valley Irrigation and Drainage District
City of Bullhead City
City of Lake Havasu City
City of Mesa
City of Somerton
City of Yuma
Electrical District No. 3, Pinal County, Arizona
Golden Shores Water Conservation District
Mohave County Water Authority
Mohave Valley Irrigation and Drainage District
Mohave Water Conservation District
North Gila Valley Irrigation and Drainage District
Town of Fredonia
Town of Thatcher
Town of Wickenburg
Salt River Project Agricultural Improvement and Power District
Unit "B" Irrigation and Drainage District
Wellton-Mohawk Irrigation and Drainage District
Yuma County Water Users' Association
Yuma Irrigation District
Yuma Mesa Irrigation and Drainage District

Other Interested Parties Participant Group

QuadState County Government Coalition
Desert Wildlife Unlimited

California Participant Group

California Department of Fish and Game
City of Needles
Coachella Valley Water District
Colorado River Board of California
Bard Water District
Imperial Irrigation District
Los Angeles Department of Water and Power
Palo Verde Irrigation District
San Diego County Water Authority
Southern California Edison Company
Southern California Public Power Authority
The Metropolitan Water District of Southern California

Nevada Participant Group

Colorado River Commission of Nevada
Nevada Department of Wildlife
Southern Nevada Water Authority
Colorado River Commission Power Users
Basic Water Company

Native American Participant Group

Hualapai Tribe
Colorado River Indian Tribes
The Cocopah Indian Tribe

Conservation Participant Group

Ducks Unlimited
Lower Colorado River RC&D Area, Inc.

Species Accounts for the Lower Colorado River Multi-Species Conservation Program

Lower Colorado River
Multi-Species Conservation Program
Bureau of Reclamation
Lower Colorado Region
Boulder City, Nevada
http://www.lcrmscp.gov

September 2008

CONTENTS

INTRODUCTION 5

THREATENED AND ENDANGERED SPECIES 7
YUMA CLAPPER RAIL 8
SOUTHWESTERN WILLOW FLYCATCHER 22
DESERT TORTOISE MOHAVE POPULATION 47
BONYTAIL 65
RAZORBACK SUCKER 74

OTHER COVERED SPECIES 97
WESTERN RED BAT 98
WESTERN YELLOW BAT 102
COLORADO RIVER COTTON RAT 114
YUMA HISPID COTTON RAT 120
LEAST BITTERN 126
CALIFORNIA BLACK RAIL 137
YELLOW-BILLED CUCKOO 150
ELF OWL 168
GILDED FLICKER 188
GILA WOODPECKER 198
VERMILION FLYCATCHER 208
ARIZONA BELL'S VIREO 218
SONORAN YELLOW WARBLER 226
SUMMER TANAGER 235
FLAT-TAILED HORNED LIZARD 243
RELICT LEOPARD FROG 264
FLANNELMOUTH SUCKER 278
MACNEILL'S SOOTYWING SKIPPER 290
STICKY BUCKWHEAT 294
THREECORNER MILKVETCH 297

EVALUATION SPECIES 300
DESERT POCKET MOUSE 301
CALIFORNIA LEAF-NOSED BAT 306
PALE TOWNSEND'S BIG-EARED BAT 315
COLORADO RIVER TOAD 330
LOWLAND LEOPARD FROG 343

INTRODUCTION

The Lower Colorado River Multi-Species Conservation Program (LCR MSCP) is a multi-stakeholder Federal and non-Federal partnership responding to the need to balance the use of lower Colorado River (LCR) water resources and the conservation of native species and their habitats in compliance with the Endangered Species Act. This is a long-term (50-year) plan to conserve at least 26 species along the LCR from Lake Mead to the Southerly International Boundary with Mexico through the implementation of a Habitat Conservation Plan (HCP)[1]. Most of the covered species are state and/or Federally listed threatened and endangered species. The Bureau of Reclamation (Reclamation) is the entity responsible for implementing the LCR MSCP over the 50-year term of the program. A Steering Committee, currently consisting of 54 entities, has been formed as described in the LCR MSCP Funding and Management Agreement (FMA), to provide input and oversight functions in support of LCR MSCP implementation.

Reclamation has developed species accounts for 22 covered species and 5 evaluation species listed in the HCP that utilize terrestrial, marsh, and riparian habitats. Bio-West, Inc., developed four species accounts for the covered native fish species (Bio-West, Inc. 2005)[2]. A species account was not developed for humpback chub as there is neither critical habitat nor occupied habitat for this species within the LCR MSCP program area.

These species accounts were based on extensive literature searches for each species and include the latest and best scientific information. These accounts include current knowledge about each species' legal status, life history, distribution, habitat requirements, behavior, and LCR MSCP Conservation Measures as it relates to the creation and management of their habitats. Only the specific Conservation Measures for each particular species were included. For a complete listing of LCR MSCP Conservation Measures, please refer to either the LCR MSCP HCP[1] or the Lower Colorado River Multi-Species Conservation Program Draft Final Science Strategy[3.]

These species accounts were developed to quantify existing knowledge for each species and to identify information gaps that, if addressed, would better inform the creation and management of covered species habitats, enabling the successful completion of Conservation Measures. LCR MSCP research and monitoring data needs have been identified for each covered and evaluation species, where appropriate. These needs will be prioritized in a 5-year plan and will be completed according to importance, urgency, and cost. Other potential research and monitoring opportunities, either identified through this process or by other scientists or conservation programs, that are outside of the scope and purpose of the LCR MSCP have also been listed to further non-LCR MSCP conservation activities.

Species accounts will be periodically updated, when appropriate, as new information is collected through monitoring and research conducted by Reclamation and others through the adaptive management process. For more information regarding the adaptive management process, refer to the Lower Colorado River Multi-Species Conservation Program Draft Final Science Strategy[3].

1. Lower Colorado River Multi-Species Conservation Program. 2004. Lower Colorado River Multi-Species Conservation Program, Volume II: Habitat Conservation Plan. Final. December 17. Available at: http://www.lcrmscp.gov.
2. Bio-West, Inc. 2005. Colorado River Backwaters Enhancement, Species Profiles Report. Submitted to Bureau of Reclamation, Lower Colorado Region, Boulder City, NV. Logan, UT. 102 p.
3. Lower Colorado River Multi-Species Conservation Program. 2006. Lower Colorado River Multi-Species Conservation Program Draft Final Science Strategy. Bureau of Reclamation, Lower Colorado Region, Boulder City, NV. Available at http://www.lcrmscp.gov.

THREATENED AND ENDANGERED SPECIES

YUMA CLAPPER RAIL
(Rallus longirostris yumanensis)

Introduction

Clapper rails (*Rallus longirostris*) are found from North America to South America and are classified into three groups: *obsoletus*, *crepitans*, and *longirostris*. The Yuma clapper rail (*Rallus longirostris yumanensis* Dickey) is one of four subspecies of the *obsoletus* group (Eddleman and Conway 1998). It was initially designated as a separate species, *Rallus yumanensis* Dickey (Bent 1926). The four species of clapper rails found along the west coast of North America, *R. obsoletus*, *R. levipes*, *R. beldingi*, and *R. yumanensis*, were later reclassified into subspecies (Van Rossem, 1929). The clapper rail primarily inhabits salt marshes and mangrove swamps throughout its range; the Yuma clapper rail inhabits freshwater marshes in the southwestern United States and northern Mexico (Eddleman and Conway 1998, Hinojosa-Huerta *et al.* 2001). It is distinguished by paler, duller underparts and grayish edging of dorsal feathers. The cheeks and postoculars are bluish or ashy gray (Eddleman and Conway 1998).

Legal Status

The Yuma clapper rail was listed as endangered on 11 March 1967 by the Secretary of Interior, pursuant to the Endangered Species Act of 1966 (U.S. Dept. of Interior 1968). California originally listed the Yuma clapper rail as endangered in 1971; relisted it as rare in 1978, and currently lists it as threatened (California Dept. of Fish and Game 2006). In 1978, Arizona classified the Yuma clapper rail as a species of special concern, similar to the Federal status of endangered (Arizona Game and Fish Dept. 2006). Nevada classifies the Yuma clapper rail as endangered as per Nevada Administrative Code 503.050. It is listed as threatened in Mexico (Hinojosa-Huerta *et al.* 2004).

Distribution

Historical Range

Grinnell (1914) did not encounter the rail during an expedition/exploration from Needles, California, to Yuma, Arizona, in 1910 (Rosenberg *et al.* 1991). The Yuma clapper rail was found along the lower Colorado River after constructions of dams and the subsequent creation of marsh habitat (Ohmart and Smith 1973). The species was first described in 1923 from one of three rails collected in 1921 near Laguna Dam, north of Yuma, Arizona (Dickey 1923). Naturalists sighted Yuma clapper rails farther north several years after Parker, Imperial, and Headgate Rock dams were completed in 1938, 1939, and 1942, respectively (Monson 1964, Phillips *et al.* 1964, Welch 1966, Ohmart and Smith 1973). Clapper rails were observed at the Salton Sea in 1931 and confirmed as Yuma clapper rails in 1940 (Moffitt 1932, Abbot 1940).

Current Range

On the lower Colorado River, this species is currently found in scattered marshes from the Colorado River Delta in Mexico, to Topock Marsh at Havasu National Wildlife Refuge (NWR), near Needles, California (Hinojosa-Huerta *et al.* 2001, Wise-Gervais 2005). Previously, the northern limit on the lower Colorado River was Laughlin Bay, Nevada (Rosenberg *et al.* 1991). The species' range now stretches north to the Virgin River and Beaver Dam Wash, near Littlefield, Arizona, and Mesquite, Nevada, the Muddy River near Overton, Nevada, and the Las Vegas Wash near Las Vegas, Nevada (McKernan and Braden 2001, Rathbun and Braden 2003). The Yuma clapper rail is also found east of the Colorado River along portions of the Gila, Salt, and Bill Williams river drainages and several other locations in central and southwestern Arizona (Arizona Game and Fish Dept. 2006). Significant populations are also found in marshes at the south end of the Salton Sea (Eddleman and Conway 1998, Patten *et al.* 2003, U.S. Fish & Wildlife Service 2005). Surveys in the Colorado River Delta in Mexico determined that the majority of Yuma clapper rails are in the Ciénega de Santa Clara, the largest marsh wetland in the delta (Hinojosa-Huerta *et al.* 2001). Clapper rails present in mangrove marshes along the west coast of Mexico may also be *yumanensis* (Eddleman and Conway 1998).

Populations within LCR MSCP Boundaries

Significant populations of Yuma clapper rail are found within the LCR MSCP boundaries in reaches 3 through 6. An analysis of survey data from 1995 to 2005 showed that between 35% and 55% of Yuma clapper rails detected in the United States were within the LCR MSCP boundaries (U.S. Fish & Wildlife Service 2005, Table 1). The majority of Yuma clapper rails located in Reach 3 were in Topock Marsh and Topock Gorge; a small population was in the marshes of the Bill Williams Delta. In Reach 4, the Cibola NWR provides habitat for almost all of the Yuma clapper rails detected during surveys. In Reach 5, rails were detected in Imperial NWR, Picacho State Recreation Area, and between Martinez Lake and Imperial Dam. In Reach 6, the majority of rails were located between Imperial Dam and Laguna Dam. On average, the percentage of Yuma clapper rails detected within the LCR MSCP boundaries were: Reach 3-27%, Reach 4-20%, Reach 5-24%, Reach 6-27%, and Reach 7-2%.

Table 1. Yuma clapper rail survey data (USFWS 2005). NS indicates no survey.

Survey Yr	**1995**	**1996**	**1997**	**1998**	**1999**	**2000**	**2001**	**2002**	**2003**	**2004**	**2005**
Reach 1	NS	NS	NS	NS	NS	NS	NS	NS	NS	NS	NS
Reach 2	0	0	0	0	1	0	NS	0	0	NS	NS
Reach 3	83	86	76	85	55	84	83	56	101	132	121
Reach 4	110	71	43	62	91	49	40	63	63	54	82
Reach 5	146	160	141	57	61	34	39	69	67	49	62
Reach 6	102	113	138	65	93	90	55	61	119	68	47
Reach 7	4	17	6	NS	0	NS	NS	3	NS	NS	9
Total	445	447	404	269	301	257	217	252	350	303	321
US Total	900	834	814	579	543	503	533	639	851	863	885
MSCP %	49.4	53.6	49.6	46.4	55.4	48.2	40.7	39.4	41.1	35.1	36.2

Life History

General Description

The Yuma clapper rail is a large, gray brown to dull cinnamon rail, with a slightly decurved bill, laterally compressed body, and long legs and toes relative to the body (Eddleman and Conway 1998). It is one of the smaller subspecies of clapper rails (Todd 1986). The total length for an adult clapper rail is 32-41 cm, with mass ranging from 160 to 400 g. Males are typically 20% larger than females (Eddleman and Conway 1998). *Yumanensis* specimens (n = 18) collected between Topock Marsh and the Colorado River Delta in 1971 had an average weight of 253 g, with males (n = 12) averaging 266.8 g and females (n = 6) averaging 226.2 g (Todd 1986). The Yuma clapper rail is the largest rail found along the lower Colorado River (Phillips *et al.* 1964). Large *yumanensis* males can stand 20 to 23 cm tall (Todd 1986). The Virginia rail is similar in shape but smaller at 22 to 27 cm in length, and has more red on the bill and more gray on the cheeks (Eddleman and Conway 1998).

Plumage is similar in both sexes. Males are somewhat brighter, although the sexes cannot be reliably separated in the field. Their upper mandible is darkish gray, diffusing into an orange base (Todd 1986). The bill is brighter in males (Eddleman and Conway 1998). In both sexes, the head has a grayish-brown forehead and crown. The side of the head, behind and below the eye, is gray. The eyelid is white, giving the appearance of a lower white eye-ring. The iris is dark brownish-orange. A white stripe extends from above the eye to the upper mandible; the posterior eye-stripe is indistinct (Todd 1986). The chin and throat are white (Dickey 1923). Upper body surfaces, including the back, scapulars, rump, and upper wing coverts, are patterned with light gray and dark brown. Brown becomes dominant towards the rump and distally on wings (Todd 1986). Primaries, primary coverts, and secondaries were described by Dickey (1923) as mummy brown. The bend of the wing is whitish. The breast is a subdued orange to burnt orange that in the breeding male becomes a brick orange. Flanks and underside are dark gray with vertical white stripes that give a barred effect. The tail is dark brown above and white below, and undertail coverts are white. The outside of the tibia is light grayish-brown with the unfeathered portions of the leg and foot a darkly tanned orange-flesh tone (Todd 1986).

The downy young are black, with black legs, and are very similar to the young of Virginia rail (*Rallus limicola)* (Eddleman and Conway 1998, Peterson 1990).

Todd (1986) and Eddleman (1989) suggested that molt patterns were similar between the Yuma clapper rail and other clapper rails. Prejuvenile molt is a complete molt, with plumage acquired in the first 6 to 8 weeks. Juvenile feathers start to appear the fourth week after hatching. Plumage begins to be replaced by first basic plumage after completion of prejuvenal molt. The definite prebasic molt for adults is complete and occurs after breeding. During this time, remiges and rectrices are lost and adult birds are flightless. Duration of the flightless period for Yuma clapper rails is 3.5 weeks (Eddleman and Conway (1998). This flightless period can occur through mid-September (Eddleman 1989).

Vocalization

Yuma clapper rails are normally heard rather than seen. In 2004, surveys conducted in Topock Gorge, Havasu NWR, encountered 177 rails audibly, with only 4 seen (USBR 2005). Most of the calls are related to territoriality and breeding behavior (Massey and Zembal 1987).

Tomlinson and Todd (1973) described seven calls for the Yuma clapper rail: kek, agitated kek, clatter, purr, agitated purr, hoo, and kek-burr. An additional five vocalizations and calls were heard during a study along the lower Colorado River: kek-hurrah, wheet, burp, kak, and rack (Eddleman 1989). Tomlinson and Todd (1973) identified the kek and clatter calls of Yuma clapper rails as identical to those given by eastern clapper rails (*R. l.* spp.). Massey and Zembal (1987) described eight calls for the light-footed clapper rail (*R. l. levipes)*. They stated, “All calls are variants on a single note; differences in sounds are due to changes in pitch, length of notes and of intervals between notes, and intensity”. The most common calls heard along the lower Colorado River are listed below.

> *Kek*. The simplest of the calls, one short note repeated many times (Massey and Zembal 1987). It is given by unmated males (Meanly 1985, Massey and Zembal 1987); Eddleman (1989) found no evidence of use by females. It is the first persistent vocalization heard annually, beginning in February, peaking in late March to late April and heard less persistently through July. The Kek call ceases when the male is mated (Eddleman 1989, Eddleman and Conway 1998).
>
> *Clatter*. Described as clapper, the common name of the rail is derived from the sound (Choate 1985, Massey and Zembal 1987, Eddleman and Conway 1998). It is the primary vocalization given by paired birds and is usually given in unison. It is heard starting in February and more commonly in April through June (Eddleman 1989). Calls between the sexes are indistinguishable (Massey and Zembal 1987). The call may function as a territorial defense (Eddleman and Conway 1998).
>
> *Kek-burr*. It is the primary advertising call of the female (Zembal and Massey 1985). It is used to attract males and may be given by unmated females, females who have lost their mates, or females calling to straying males (Zembal and Massey 1985). It is only heard during the breeding season (Massey and Zembal 1987).
>
> *Kek-hurrah*. This call is not specific to either sex and may be an abbreviated form of the clatter (Eddleman and Conway 1998).
>
> *Agitated kek.* This call is given when a rail is disturbed or distressed (Todd 1986, Massey and Zembal 1987).

Breeding

Age at first breeding is unknown but presumed to be 1 year (Eddleman and Conway 1998). In Arizona, males begin advertising in February and pair formation begins shortly afterward (Eddleman and Conway 1998). Nests were recorded in Arizona on 13 March (Eddleman 1989). Records from the University of California's Museum of Vertebrate Zoology and nest cards from the Cornell Laboratory of Ornithology indicate the mean date for first brood in southwest Arizona and southeast California is 1 May ± 24.8 days (Eddleman and Conway 1998). Mean clutch size is 6.8, ranging from 6 to 8 eggs (Eddleman 1989). In southwest Arizona, egg laying and caring for young begins in mid-March and occurs through early September (Eddleman and Conway 1998). Both sexes incubate nests, typically females in the day and males at night. Incubation period at seven nests in Arizona was 23-28 days (Eddleman 1989).

Five nests found at the Salton Sea were either constructed of black sticks with a few dead leaves on them, or fine stems with dry blossoms on them. Two of the nests were found on small mud hummocks; the other three nests were in crotches of small shrubs, just above water in dense cattail and tamarisk habitat (Abbott 1940). In Arizona, Eddleman (1989) measured nest diameters ranging between 22 and 32 cm, nest depth ranging between 0 and 8 cm, and nest height between 6 and 92 cm. Nests were found in the base of living clumps of bulrush (Cyperaceae), cattail (*Typha*), or a saltcedar (Tamarisk), under wind-thrown bullwhip bulrush (*Juncus californicus*), and within or on top of dead cattail remaining from previous years. Half of these nests lacked ramps (elevated entrances from substrate or water surface to nest rim) and all lacked canopies (Eddleman 1989). Yuma clapper rail nests were found near shore, in shallow water, and in marsh interiors over deep (>1 m) water (Eddleman 1989, Conway *et al.* 1993).

Clapper rail young are precocial (Meanly 1985). First-hatched chicks are led from the nest by one parent, while the remaining parent continues incubation of new hatched chicks and remaining eggs (Kozicky and Schmidt 1949, Adams and Quay 1958). Chicks are fed fragments of prey eaten by adults (Adams and Quay 1958, Zembal and Fancher 1988). Young rails learn foraging strategies from adults but may be fed, in part, by adults until the age of 6 weeks (Zembal and Fancer 1988). Parental care extends to the fifth or sixth week (Adams and Quay 1958, Zembal and Fancher 1988) but brooding may continue until eighth or tenth week (MacNamara and Udell 1970 *in* Eddleman and Conway 1998). Young are able to fly after 10 weeks and become indistinguishable from adults. There is no apparent association with brood mates or parents after fledging (Eddleman and Conway 1998).

Adams and Quay (1958) observed young clapper rails, aged 9-10 weeks, making calls like those of adults, while Meanly (1985) observed captive clapper rails, 6 months old, making the primary advertising call of an adult.

Diet

Clapper rails are sight feeders, gleaning the surface, making shallow and sometimes deep probes, gleaning below the water surface, moving at times erratically in search of prey, and at other times moving slowly and deliberately (Simmons 1914, Williams 1929, Meanly 1985, Todd 1986, Zembal and Fancher 1988). In Arizona, the Yuma clapper rail forages at sites with high mean coverage by surface water, low stem density relative to other sites in marshes, and moderate water depth (about 7.5 cm) (Eddleman 1989, Conway *et al.* 1993).

Information on the feeding habits of the Yuma clapper rail is somewhat limited. Ohmart and Tomlinson (1977) collected Yuma clapper rail specimens from Topock Marsh to Imperial Reservoir, the confluence of the Gila River and Colorado River, and the Colorado River Delta in Sonora, Mexico. In rails from Topock Marsh to Imperial Reservoir, crayfish (*Procambarus clarki* and *Orconectes* spp.) (95%) were the dominant food item, followed by weevils (Curculionidae), unidentified beetles (Coleoptera), spiders, damselfly nymphs (Zygoptera), grasshoppers (Orthoptera), insect eggs, ground beetles (Carabidae), plant seeds, an unidentified mammal bone, and an introduced freshwater clam (*Corbicula* spp.). The rails at the confluence of the Gila River and the Colorado River were utilizing *Corbicula* (50%), isopods (48.5%), and unidentified insects. Rails at the Colorado River Delta in Mexico were utilizing water beetles (Hydrophilidae) (56.5%), unidentified fish (32%), leeches, plant matter (seeds and twigs), damselfly nymphs, dragonfly nymphs (Anisoptera), and shrimp (*Palaemonidae* spp.). Specimens of two other species of clapper rails, Sonora clapper rail (*R. l. rhizophorae*) and San Blas clapper rail (*R. l. nayaritensis*), were obtained in the mangrove swamps in the states of Sonora, Sinaloa, and Nayarit. Omart and Tomlinson (1977) observed that, despite a great abundance and variety of invertebrate food species available to the rails in the mangrove swamps, crabs (87% and 98%) were selected in preference to the other available foods. They concluded that, within the limits of their investigations, “Clapper Rails were selective, opportunistic, or limited in the variety of foods eaten depending upon habitat type”.

Todd (1986) observed Yuma clapper rails capturing small fish and tadpoles and gleaning invertebrates from algae covered parts of cattails under the water surface. Eddleman (1989) looked at food habits and prey availability for Yuma clapper rail at Crystal Beach in Topock Gorge and at Mittry Lake. Stomach contents were collected from two mortalities during the study. A male from Mittry Lake contained two crayfish and three freshwater prawns (*Palaemonetes paludosus*). Another male from Crystal Beach had an earwig (Demaptera) in its esophagus. Regurgitated cast pellets were collected at both sites and only contained the remains of crayfish. Active and passive traps were set in cattail, bullwhip bulrush, and saltcedar habitats in Crystal Beach and traps were set in cattail and bullwhip bulrush at Mittry Lake. Crayfish and freshwater prawns comprised most of the potential prey items, with the next item of importance being water boatmen (Corixidae). At Mittry Lake, the potential prey was more diverse. They were mosquitofish (*Gambusia affinis*), freshwater prawns, crayfish, bullfrogs *(Rana catesbiana*) (adults and tadpoles), sailfin mollies (*Poecilia latipinna*), water beetles, predaceous diving beetles (Dytiscidae), dragonfly nymphs (Aeshnidae, Libellulidae), and water boatmen.

Habitat

Yuma clapper rails are found in a variety of marsh types that are dominated by emergent plants, including southern cattail (*Typha domingensis*), bullwhip bulrush, three-square bulrush (*Scirpus olneyi*), and sedges (Cyperaceae) (Todd 1986). The presence of emergent cover, not the plant species or marsh size, is an important trait of habitat (Anderson and Ohmart 1985). In Arizona, habitat studies determined that sites with high coverage by surface water, low stem density, and moderate water depth were used for foraging during the nesting season, while sites with high stem density and shallower water near shorelines were used for nesting (Conway 1990, Conway *et al.* 1993). Habitat used in early winter (November-December) has lower emergent stem density, basal coverage, and ground coverage; less distance to water; greater overhead coverage by vegetation, distance to adjacent uplands, distance to vegetative edges, water depth and water coverage; and taller emergent plants than do randomly selected sites (Eddleman 1989, Conway *et al.* 1993).

Low stem densities and little residual vegetation are features of year-round rail habitat (Conway *et al.* 1993). Eddleman and Conway (1998) also described the ideal habitat as being a mosaic of emergent plant stands of different ages, interspersed with shallow pools of open water.

Estimates of home range at Mittry Lake were 24 ha in late winter (January-February), 8.3 ha during early breeding (March-April), 6.7 ha during late breeding (May-July), 14.7 ha during post breeding (August-October), and 8.8 ha in early winter (November-December) (Conway 1990). Data was collected by attaching radio transmitters to rails and monitoring their movements during 1985-1987 (Conway *et al.* 1993). Previous estimates of home range varied between 0.12 ha and 3.9 ha during the breeding season and are based on vocalizations, which may not be an effective method of determining area use size of Yuma clapper rails (Todd 1986, Tomlinson and Todd 1973, Conway *et al.* 1993).

Migration

Data is inconclusive concerning the migration of the Yuma clapper rail. Investigations that used response to broadcast calls as an indication of presence concluded that there was probable migration of Yuma clapper rail from the United States (Tomlinson and Todd 1973, Todd 1986). Wise-Gervais (2005) believes that Gila River populations are migratory. However, more recent studies have concluded that, although Yuma clapper rails increase movement and home range size during the winter, individuals do utilize the lower Colorado River all year. Changes in detection rates may influence broadcast call surveys conducted outside the breeding season (Conway *et al.* 1993). Studies using radio telemetry to monitor rails year-round could not confirm migration (Eddleman 1989, Conway 1990).

Threats

Predation is the main mortality factor for adult Yuma clapper rails (Eddleman 1989). Coyotes (*Canis latrans*), raccoons (*Procyon lotor*), and raptors such as northern harrier (*Circus cyaneus*), great horned owl (*Bubo virginicus*), and Harris' hawk (*Parabuteo unicinctus*), have been documented as predators of the Yuma clapper rail (Eddleman

1989). Striped skunks (*Mephitis mephitis*) are a potential predator of adult rails, and bullfrogs (*Rana catesbeiana*), black bass (*Micropterus salmoides*), softshell turtle (*Trionyx* spp.), and common king snakes (*Lampropeltis getulus*) are potential predators of young rails and eggs (Todd 1986).

Fire during the breeding season (mid-March to early September) can cause loss of eggs, young, and some adults (Todd 1986). After breeding, adults go through a prebasic molt, lose their tail and flight feathers, and remain flightless for 3.5 weeks (Eddleman and Conway 1998). This flightless period can occur through mid-September (Eddleman 1989), and fires during this time could severely impact rails.

Degradation of habitat is thought to be a factor contributing to declines in rail populations (Conway and Nadeau 2005). The lack of stochastic events that would scour and rejuvenate wetlands has allowed encroachment by woody vegetation and buildup of large amounts of decadent vegetation (Conway and Nadeau 2005). Yuma clapper rails select for high stem density during the nesting period; however, low stem densities and limited residual vegetation are features of year-round rail habitat (Conway 1990, Conway *et al.* 1993).

Selenium occurs naturally within the lower Colorado River Basin. Although mortality or reproductive impairment have not been documented in Yuma clapper rail populations along the lower Colorado River, concentrations of selenium in the Yuma clapper rail's food chain may be within the range that could cause adverse effects on reproduction (Eddleman 1989, King *et al.* 2000). One recent study indicated that selenium concentrations doubled over a 10-year time period (King *et al.* 2000).

Drying or drainage of managed wetlands can result in nest abandonment (Johnson and Dinsmore 1985, Bennet and Ohmart 1978 *in* Eddleman 1989). Rising water levels force rails to higher ground where they become predisposed to predation (Eddleman 1989). Prolonged higher than usual water levels can cause abandonment of territories (Smith 1975).

Yuma clapper rails are threatened by river management activities that are detrimental to marsh formation, such as dredging, channelization, bank stabilization, and other flood control measures (LCR MSCP 2004a).

LCR MSCP Conservation Measures

CLRA1—Create 512 acres (229 ha) of Yuma clapper rail habitat. Create and manage 512 acres of marsh to provide Yuma clapper rail habitat. This created habitat will also provide habitat for the least bittern (*Ixobrychus exilis*) and the California black rail (*Laterallus jamaicensis coturniculus*). Habitat will be created in patches as large as possible but will not be created in patches smaller than 5 acres (2 ha). Smaller patches are likely to support isolated nesting pairs and be within the range of habitat patch sizes used by the species for foraging and dispersal. Larger patches would be expected to support multiple nesting pairs. Additional Yuma clapper rail habitat may be provided by marsh vegetation that becomes established along margins of the 360 acres (146 ha) of backwaters that will be created in reaches 3-6. These small patches of habitat would provide cover for dispersing rails, thereby facilitating linkages between existing breeding populations and the colonization of created habitats.

Yuma clapper rail habitat will be created and maintained as described in section 5.4.3.3. Marshes created to provide Yuma clapper rail habitat will be designed and managed to provide an integrated mosaic of wetland vegetation types, water depths, and open water areas. Within this mosaic of marsh conditions, Yuma clapper rail habitat will generally be provided by patches of bulrush and cattails interspersed with small patches of open water with water levels maintained at depths appropriate for this species (no more than 12 inches, 30.5 cm).

CLRA2—Maintain existing important Yuma clapper rail habitat areas. The Applicants, under agreements with cooperating land management agencies, will provide funding to those agencies to maintain a portion of existing Yuma clapper rail habitat within the LCR MSCP planning area (Section 5.4.2). Maintaining important existing habitat areas is necessary to ensure the continued existence of Yuma clapper rails in the LCR MSCP planning area, provide for the production of individuals that could disperse to and nest in LCR MSCP-created habitat, and support future recovery of the species. Habitat maintenance would likely be undertaken in conjunction with the maintenance of existing California black rail habitat (LCR MSCP 2004b).

LCR MSCP Research and Monitoring Needs

The Yuma clapper rail was studied intensely during the 1980s as a result of the Federal Endangered Species Act process. Data were collected on clapper rail biology and demographics. However, several additional data needs have been identified that will enable the conservation measures to be accomplished. LCR MSCP research needs include:

- Conduct diet studies to determine whether the Yuma clapper rail is utilizing all the potential prey that is available. Eddleman (1989) found a wide variety of prey available to rails at Mittry Lake; however, it is unknown if the rails take advantage of available prey items.

- Monitor selenium levels in the Yuma clapper rail and available prey to determine current levels of selenium concentration and to determine whether concentrations are increasing over time. Conduct studies to determine whether selenium levels affect reproductive success.

- Conduct studies to determine Yuma clapper rail movement within habitat and migration from its habitat during the winter months. Rails monitored by radio telemetry at Mittry Lake did not leave in the winter months (Conway 1990);

however, the percent of clapper rails that do migrate during the winter to other sites is unknown.

- Determine the effects of water fluctuation on nesting Yuma clapper rails. Water levels at Topock Gorge can fluctuate as much as 4-5ft (1.2-1.5 m) during March through May (USBR 2004). Jackson (1983) observed a pair of clapper rails in Mississippi respond to high water by building their nest higher; the depth of the nest was later measured at 19.5 cm. Eddleman (1989) found one nest 92 cm above the substrate and presumed it was built at the level of the water earlier in the year. Eddleman *et al.* (1988) recommended that manipulation of water levels on diked marsh units should be examined as a possible management tool for the Yuma clapper rail.

Other Potential Research and Monitoring Opportunities

The largest populations of Yuma clapper rail exist outside of the LCR MSCP boundary, specifically in Mexico. The Yuma clapper rail recovery plan details actions that include monitoring and research needs in Mexico, including breeding surveys and winter habitat studies (USFWS 1983).

Literature Cited

Adams, D. A., and T. L. Quay. 1958. Ecology of the clapper rail in southeastern North Carolina. Journal of Wildlife Management 22:149-156.

Abbott, C. G. 1940. Notes from the Salton Sea, California. Condor 42:264-265.

Anderson, B. W., and R. D. Ohmart. 1985. Habitat use by clapper rails in the lower Colorado River Valley. Condor 87:116-126.

Arizona Game and Fish Department. 2006. Yuma Clapper Rail. Unpublished abstract complied and edited by the Heritage Data Management System, Arizona Game and Fish Department, Phoenix, AZ. 11 p.

Bent, A. C. 1926. Life histories of the North American marsh birds. U.S. Natural Museum Bulletin 135. Dover Publications, New York. 385 p.

Bureau of Reclamation. 2004. Yuma clapper rail survey along lower Colorado River at Topock Gorge, 1996-2003, Final Report. Lower Colorado Region, Natural Resources Office. 23 p.

Bureau of Reclamation. 2005. Yuma clapper rail survey along lower Colorado River at Topock Gorge, 2004, Final Report. Lower Colorado River Multi-species Conservation Program. 19 p.

California Department of Fish and Game. 2006. List of the state and federally endangered and threatened animals of California. California Department of Fish and Game, Sacramento. 12 p.

Choate, E. A. 1985. The Dictionary of American Bird Names, Revised Edition. The Harvard Common Press. Boston. 226 p.

Conway, C. J. 1990. Seasonal changes in movements and habitat use by three sympatric species of rails. M.S. Thesis, University of Wyoming, Laramie.

Conway, C. J., W. R. Eddlemen, S. H. Anderson, and L. R. Hanebury. 1993. Seasonal changes in Yuma Clapper Rail vocalization rate and habitat use. Journal of Wildlife Management 56:282-290.

Conway, C. J., and C. P. Nadeau. 2005. Effects of fire on Yuma clapper rails and California black rails, 2004 annual report. Wildlife Report Number 2005-01. Arizona Cooperative Fish and Wildlife Research Unit, Tucson, AZ. 14 p.

Dickey, D. R. 1923. Description of a new clapper rail from the Colorado River Valley. Auk 40:90-94.

Eddleman, W. R., F. L. Knopf, B. Meanly, F. A. Reid, and R. Zembal. 1988. Conservation of North American ralllids. Wilson Bulletin 100:458-475.

Eddleman, W. R. 1989. Biology of the Yuma clapper rail in the southwestern U.S. and northwestern Mexico. Final Report, Intra-Agency Agreement No. 4-AA-30-02060, Bureau of Reclamation, Yuma Project Office, Yuma, AZ. 127 p.

Eddleman, W. R., and C. J. Conway. 1998. Clapper rail (*Rallus longirostris yumanensis*). *In* The Birds of North America, No. 340 (A. Poole and F. Gill eds.) The Birds of North America, Inc., Philadelphia, PA.

Grinnell, J. 1914. An account of the mammals and birds of the lower Colorado Valley. University of California Publications in Zoology 12:51-294.

Hinojosa-Huerta, O., S. DeStefano, and W. W. Shaw. 2001. Distribution and abundance of the Yuma clapper rail (*Rallus longirostris yumanensis*) in the Colorado River Celta, México. Journal of Arid Environments 49:171-182.

Hinojosa-Huerta, O., H. Iturribarría-Rojas, Y. Carrillo-Guerrero, M. de la Garza-Treviño, and E. Zamora-Hernádez. 2004. Bird conservation plan for the Colorado River Delta. Pronatua Noroeste, Dirección de Conservación Sonora. San Luis Río Colorado, Sonora, México.

Jackson, J. A. 1983. Adaptive response of nesting clapper rails to unusually high water. Wilson Bulletin 95:308-309.

Johnson, R. R., and J. J. Dinsmore. 1985. Broodrearing and postbreeding habitat use by Virginia Rails and Soras. Wilson Bull. 97: 551-554.

King, K. A., A. L. Velasco, J. Garcia-Hernandez, B. J. Zaun, J. Record, and J. Wesley. 2000. Contaminants in potential prey of the Yuma clapper rail: Arizona and California, USA, and Sonora and Baja, Mexico, 1998-1999. U.S. Fish and Wildlife Service, Ecological Services Field Office, Phoenix, AZ. 22 p.

Kozicky, E. L., and F. W. Schmidt. 1949. Nesting habits of the clapper rail in New Jersey. Auk 66:355-364.

Lower Colorado River Multi-Species Conservation Program. 2004a. Lower Colorado River Multi-Species Conservation Program, Volume IV: Appendices to Volume I-III and V. Final. December 17. (J&S 00450.00) Sacramento, CA.

Lower Colorado River Multi-Species Conservation Program. 2004b. Lower Colorado River Multi-Species Conservation Program, Volume II: Habitat Conservation Plan. Final. December 17. (J&S 00450.00.) Sacramento, CA.

Massey, B. W., and R. Zembal. 1987. Vocalizations of the light-footed clapper rail. Journal of Field Ornithology 58:32-40.

McKernan, R. L., and G. T. Braden. 2001. The status of Yuma clapper rail and yellow-billed cuckoo along portions of Virgin River, Muddy River, and Las Vegas Wash, southern Nevada, 2000. San Bernardino County Museum, Redlands, CA. 19 p.

Meanly, B. 1985. The marsh hen: a natural history of the clapper rail of the Atlantic coast salt marsh. Tidewater Publications, Centreville, MD. 123 p.

Moffitt, J. 1932. Clapper rails occur on marshes of Salton Sea, California. Condor 34:137.

Monson, G. 1964. Recent notes from the lower Colorado River Valley of Arizona and California. Condor 51:262-265.

Ohmart, R. D., and R. W. Smith. 1973. North American clapper rail (*Rallus longrirostris*) literature survey with special consideration being given to the past and current status of *yumanensis*. Bureau of Reclamation 14-06-300-2409. 40 p.

Ohmart, R. D., and R. E. Tomlinson. 1977. Foods of western clapper rails. Wilson Bulletin 89:332-336.

Patten, M. A., G. McCaskie, and P. Unitt. 2003. Birds of the Salton Sea: status, biogeography, and ecology. University of California Press, Berkeley. 363 p.

Peterson, R. T. 1990. A Field Guide to Western Birds. Houghton Mifflin Co., Boston. 432 p.

Phillips, A. J., J. Marshall, and G. Monson. 1964. The Birds of Arizona. University of Arizona Press. Tucson. 212 p.

Rathbun, M. R., and G. T. Braden. 2003. The status of Yuma clapper rail and yellow-billed cuckoo along portions of the Virgin River and Muddy River in southern Nevada for 2002. San Bernardino County Museum. Redlands, CA. 29 p.

Rosenberg, K. V., R. D. Ohmart, W. C. Hunter, and B. W. Anderson. 1991. Birds of the lower Colorado River Valley. University of Arizona Press, Tucson. 416 p.

Simmons, G. F. 1914. Notes on the Louisiana clapper rail (*Rallus crepitans saturatus*) in Texas. Auk 31:363-384.

Smith, P. M. 1975. Habitat requirements and observations of the Yuma clapper rail. M.S. Thesis, University of Arizona, Tucson.

Todd, R. L. 1986. A saltwater marsh hen in Arizona: a history of the Yuma clapper rail (*Rallus longirostris yumanensis*). Completion Report, Arizona Game and Fish Department., Federal Aid Project W-95-R, Phoenix. 290 p.

Tomlinson, R. E., and Todd R. L. 1973. Distribution of two western clapper rail races as determined by response to taped calls. Condor 75:177-183.

U.S. Fish and Wildlife Service. 1983. Yuma clapper rail recovery plan. U.S. Fish and Wildlife Service, Albuquerque, NM. 51 p.

U.S. Fish and Wildlife Service. 2005. Yuma clapper rail survey data 1995-2005: corrected version: August 25, 2005. Ecological Services Field Office, AZ. 1 p.

Welch, J. M. 1966. Southwest Region. Audubon Field Notes 20:589-591.

Williams, L. W. 1929. Notes on the feeding habits and behavior of the California clapper rail. Condor 31:52-56.

Wise-Gervais, C. 2005. Clapper rail (*Rallus longrirostris*). *In* Arizona Breeding Bird Atlas (T. E. Corman and C. Wise-Gervais, eds.). University of New Mexico Press, Albuquerque. 636 p.

Van Rossem, A. J. 1929. The status of some Pacific coast clapper rails. Condor 31:213-215.

Zembal, R., and B. W. Massey. 1985. Function of a rail mystery call. Auk 102:179-180.

Zembal, R., and J. M. Fancher. 1987. Foraging behavior and foods of the light-footed clapper rail. Condor 90:959-962.

SOUTHWESTERN WILLOW FLYCATCHER (*Empidonax traillii extimus*)

Legal Status

The southwestern willow flycatcher (SWFL) was designated by the U.S. Fish and Wildlife Service as endangered on 27 February 1995 (USFWS 1995). A final recovery plan was completed in August 2002 (USFWS 2002), and the designation of critical habitat was finalized in October 2005 (USFWS 2005). Critical habitat was previously designated on 22 July 1997 (62 FR 39129), but was rescinded by court order on 11 May 2001. This subspecies was listed under the California Endangered Species Act as endangered in 1990, and is on the list of Arizona Wildlife of Special Concern (LCR MSCP 2004).

Distribution

Historical Range

The historic breeding range of the SWFL included southern California, southern Nevada, southern Utah, Arizona, New Mexico, western Texas, southwestern Colorado, and extreme northwestern Mexico (Unitt 1987, Browning 1993, Paxton 2000, USFWS 2002). Several museum specimens of *E .t. extimus* have been collected since the early 1900s. Many of these specimens have been analyzed and those in southern California and along the lower Colorado River (LCR) have been determined to be *E. t. extimus* (Unitt 1987). Two specimens near Laguna Dam were collected in June 1930 and August 1938; both were assumed to be breeding. Other specimens were collected near Laguna Dam in May 1930; however, breeding status was undetermined. One specimen was collected along the Colorado River, 5 miles northeast of Yuma in 1910 (Grinnell 1914, Unitt 1987). Another indication of a substantial willow flycatcher population along the Colorado River is presented through evidence of 37 nests collected near Yuma, Arizona, in 1902; 33 of these nests are now at the University of Arizona (Unitt 1987). In Nevada, specimens were collected on the Colorado River, at the southern tip of the state, in May 1953 (Unitt 1987).

Current Distribution

According to the critical habitat designation for SWFL, the current occupied geographic area crosses six southwestern states including southern California, southern Nevada, southern Utah, southern Colorado, Arizona, and New Mexico, from sea level to approximately 8000 feet above sea level. Genetic studies conducted by Paxton (2000) helped to further define the boundaries. The current breeding range maps are located in both the Recovery Plan (USFWS 2002) and Critical Habitat designation (USFWS 2005) (figure 1). In general, flycatcher distribution occurs mainly in lower elevation riparian habitat, with a few patches distributed in relatively small isolated locations. According to the SWFL Recovery Plan, approximately 53% of the known SWFL population is found in 10 breeding sites range-wide, while the other 47% are distributed among approximately 100 small sites of 10 or fewer territories (USFWS 2002).

When SWFL was listed as endangered in 1995, populations were estimated at 350 territories (USFWS 2002). Through an increase in survey effort, that number has increased to over 1000 territories (Durst *et al.* 2005). Arizona Game and Fish documented 883 resident flycatchers at 483 territories in 47 sites in 2005 (English *et al.* 2006). Nevada Department of Wildlife reported a total of 18 resident flycatchers at sites not surveyed by SWCA Environmental Consultants, and an average of 9 territories for surveys from 2001 to 2005. Approximately 73 territories were documented in 2005 by SWCA Environmental Consultants along the lower Colorado River and at sites in Nevada and the lower Grand Canyon (Koronkiewicz *et al.* 2006a).

Another important aspect to the distribution of SWFL is migration routes and migration stopover habitats. This neo-tropical migrant travels between breeding areas in the United States to wintering grounds in Central and South America (USFWS 2005). Migration flyways include major river corridors and their tributaries such as the Gila River, Rio Grande River, and the lower Colorado River (Yong and Finch 1997, Moore 2005, Koronkiewicz *et al.* 2006a, English *et al.* 2006, USFWS 2005). More than 600 individual birds have been located during migration along the LCR alone, especially in areas near Yuma, Arizona (McLeod *et al.* 2005).

Wintering grounds for the willow flycatcher include portions of southern Mexico, Central America, and northern South America. Specific surveys have been conducted at sites in El Salvador, Costa Rica, Panama, Mexico, Ecuador, Nicaragua, and Guatemala (Phillips 1948, Koronkiewicz and Whitfield 1999, Koronkiewicz and Sogge 2000, Lynn and Whitfield 2002, Lynn *et al.* 2003, Nishida and Whitfield 2005). It is suspected that all subspecies may winter in similar locations. Because it is difficult to identify subspecies of willow flycatchers, specific areas where the SWFL winters are not fully known at this time.

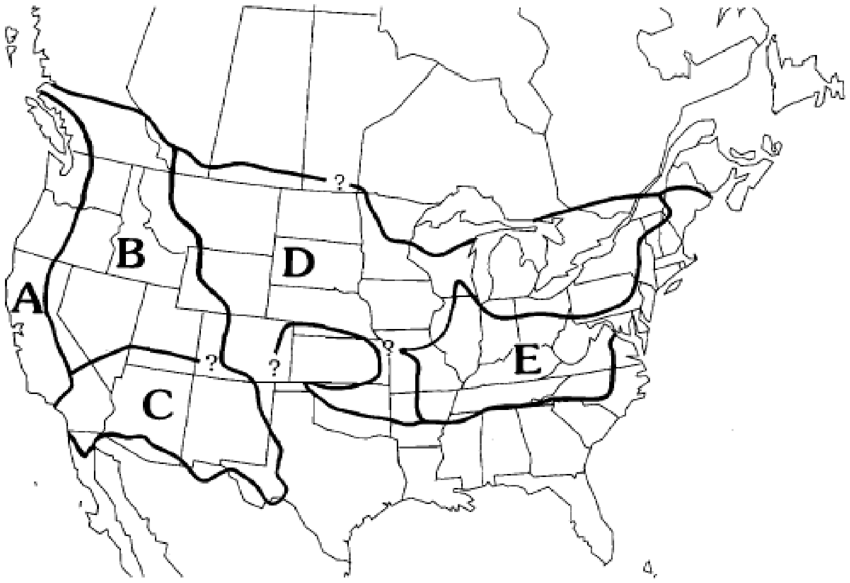


Figure 1. Approximate breeding range of subspecies of *Empidonax traillii*: A. *brewsteri*; B. *adastus*; C. *extimus*; D. *campestris*; E. *traillii*. *Empidonax alnorurn* breeds north to Alaska (Browning 1993).

Populations Within LCR MSCP Project Boundary

Presence/absence surveys, along with life history studies, have been conducted along the LCR since 1996 (McKernan and Braden 1997, 1998, 1999, 2001a, 2001b, 2002, 2006, Koronkiewicz *et al.* 2004, 2006a and McLeod 2005). Approximately 100 sites have been surveyed in an area that includes the Virgin River, Pahranagat NWR, the Grand Canyon south of Separation Canyon, and throughout the LCR from Lake Mead to the Southerly International Boundary with Mexico. These surveys indicate that the main breeding populations occur along the Virgin River from north of Mesquite, Nevada, to the Virgin River Delta with Lake Mead, at Pahranagat National Wildlife Refuge, in the Grand Canyon from Separation Canyon to the delta of Lake Mead, at Topock Marsh near Needles, California, and on the Bill Williams National Wildlife Refuge. No nests have been located south of the Bill Williams River, Arizona, in over 65 years (Unitt 1987, Koronkiewicz *et al.* 2006a).Willow flycatchers also have been detected during migration at several sites along the Colorado River, south of the Bill Williams River to the Mexico border, with over 200 detections recorded in 2003, over 600 in 2004, and over 300 in 2005 (Koronkiewicz *et al.* 2006a). Behavioral observations and timing of detections strongly suggest this section of the river is a major flyway for migrant willow flycatchers. Populations along the LCR over the past 10 years are listed in Table 1.

Table 1. Southwestern Willow Flycatcher (*Empidonax traillii extimus*) population along the lower Colorado River from 1996 to 2005.

Year	Total WIFL Detected*	SWFL Summer Resident/ Breeding
1996	202	34
1997	154	68
1998	302	113
1999	NA	133
2000	NA	135
2001	NA	218
2002	NA	142
2003	356	115
2004	793	193
2005	473	133

*Total WIFL Detected = Migratory and breeding birds detected during presence/absence surveys. Migratory birds include other subspecies than just *E. t. extimus,* and thus we can only confirm to species *E. trailii.* NA = not available.

Life History

General Description

The willow flycatcher is one of 10 subspecies in the genus *Empidonax* (meaning gnat or mosquito king), as proposed by Jean Cabanis in 1855 (Phillips *et al.* 1964, McCabe 1991, Sogge *et al.* 1997a). "Willow flycatcher" was differentiated in 1831 by John James Audubon along the Arkansas River under the name *Musicapa traillii* (McCabe 1991). The alder flycatcher *Empidonax alnorum* was separated from *E. traillii* by AOU in 1973 (AOU 1973). The southwestern willow flycatcher (*Empidonax traillii extimus*) is now recognized as one of four, or possibly five, subspecies of the willow flycatcher (Phillips 1948, Unitt 1987, Browning 1993, Paxton 2000) (Figure 1). The subspecies include *E. t. brewsteri*, located along the northwestern coast of the United States; *E. t. adastus*, located in the Great Basin and northern Rocky Mountains; *E. t. traillii*, located in the northern and eastern United States; *E. t. extimus*, located in the Southwest; and potentially *E. t. campestris* in the central United States. Some authors consider *E. t. campestris* and *E. t. traillii* as synonymous (Unitt 1987, Paxton 2000), but do acknowledge that more study is needed. These species are separated by geographic and/or environmental boundaries: *E. t. adastus* and *E. t. brewsteri* by the Cascade and Sierra Nevada mountain ranges; *E. t. adastus* and *E. t. traillii* by the Rocky Mountains; and *E. t. extimus* by distinctions in abiotic environments (although the northern boundary of *E. t. extimus* is similar to the southern portion of *E. t. adastus* habitat) (Paxton 2000).

Willow flycatcher subspecies are distinguished by subtle differences in color, morphology, and genealogy. The SWFL subspecies is generally paler in color and considered grayish olive or pale grayish green (Unitt 1987, USFWS 2002). Differences in wing formula, bill length, and wing:tail ratio also distinguish the SWFL from other subspecies (Unitt 1987). Evidence also suggests that song variations may distinguish the SWFL from other subspecies (Sedgwick 2001). Genetic structuring is highly indicative of the separation of the four subspecies, with a significant degree of separation between *E. t. extimus* and the other three more northern subspecies. This distinction also suggests greater isolation and differences in demographic history (Paxton 2000). There has been a loss in some genetic variation, which is indicative of a severe demographic bottleneck over the last 100-150 years (Paxton 2000).

The southwestern willow flycatcher is a small neotropical migrant that primarily lives along riparian corridors in dense trees and shrubs. These riparian habitats are associated with rivers, wetlands, lakes, and reservoirs; the wetlands are classified as palustrine and lacustrine forested wetlands and scrub-shrub wetlands (Cowardin *et al.* 1979). Surface water and/or saturated soil is typically present at least seasonally, and in most cases year-round, with water depths of 2-3 meters located within or adjacent to the nesting habitat (USFWS 2002).

The flycatcher is approximately 15 cm (5.75 in) long, and weighs approximately 12 g. It has a grayish-green back and wings, whitish throat, light grey-olive breast, and pale yellow belly. Two distinct wing bars are visible on the greater coverts, and an eye-ring is either absent or very faint. The upper mandible is dark, while the lower mandible is pale to yellowish (Phillips *et al.* 1964, USFWS 2002).

Recognition of the different subspecies in the field is nearly impossible, and is mainly based on differences in color and morphology, using museum specimens (Unitt 1987, Paxton 2000). The SWFL may be distinguished from other *Empidonax* species by its primary song and only on its breeding grounds after spring migration is over (Sogge *et al.* 1997a, Sogge *et al.* 1997b). The song, made by both male and female, is a sneezy "fitz-bew", and its call is a repeated "whitt". Other vocalizations for the SWFL include the "wheeo" and rolling "brrrt" notes (McCabe 1991, Sogge *et al.* 1997b, Braden and McKernan 1998, USFWS 2002). Only the "fitz-bew" song is used as a positive identification for use in presence/absence surveys (Sogge *et al.* 1997a, USFWS 2000). Migrant flycatchers often sing from tall song perches during spring migration, similar to territorial birds. Male flycatchers sing early in the breeding season and early in the nesting cycle to establish territories. Song rate declines as the season progresses, especially once mating and nesting occurs (Braden and McKernan 1998). During the initiation of breeding, primary song can usually be heard during pre-dawn chorus, sometimes throughout the day or during the evening, beginning at sunset. Short periods of pre-dawn singing may occur throughout the breeding season (Yard and Brown 2003, McCabe 1991).

Little is known of diseases and parasites within the SWFL population. McCabe (1991) reported a mite infestation in several willow flycatcher nests in Maryland, identified as *Ornithonyssus sylviarum*, the northern fowl mite. The SWFL is also known to host blood parasites such as *Hemoproteus*, *Leucocytozoon*, *Microfilaria*, *Tyrpanosoma*, and *Plasmodium* (USFWS 2002). Other parasites identified include blow fly (*Protocalliphora* spp.) and nasal mites (USFWS 2002). It is unknown what effects these parasites have on the SWFL, but McCabe (1991) noted no significant effects from the mite infestations. No studies have been conducted, nor have any of the reports from the LCR studies indicated any problems with either disease or parasites.

Breeding

The SWFL breeds across the lower southwestern United States from May through August. SWFL typically arrive on the breeding grounds between early May and early June. Males generally arrive first to set up territories, with females arriving a week or two later. Males are highly territorial and will defend their territory through counter singing and aggressive interaction. Flycatchers often clump together in one area of the habitat patch, which leads to an indication that this species is "semi-colonial". Males are usually monogamous, but polygyny does occur at approximately 10-20% (USFWS 2002, Pearson 2002). Genetic evidence suggests extra pair copulation exists by either mated or unmated males with females in neighboring territories (USFWS 2002).

Territory size varies greatly, potentially due to population density, habitat quality, and nesting stage. Territory sizes have been estimated from approximately 0.1 ha to 2.3 ha (0.25-5.7 ac) (McCabe 1991), with most territories encompassing 0.2-0.5 ha (0.5-1.2 ac) (Sogge 1995, USFWS 2002). Territories of polygynous males are usually larger than those of monogamous males. Flycatchers' home ranges are greater than their territories and can vary in size from 0.13 to 360 ha, depending on breeding status and surrounding habitat areas (Bakian and Paxton 2004, Cardinal and Paxton 2005). Home ranges are greater for non-breeding birds, and during pre- or post-breeding times (Bakian and Paxton 2004, Cardinal and Paxton 2005). During breeding, home ranges were greatly reduced and were typically less than 0.5 ha (Bakian and Paxton 2004) Territoriality is also maintained on the wintering grounds (Koronkiewicz *et al.* 2006b).

Multi-year color banding studies have shown high site fidelity among after-second-year birds returning to former breeding patches (McKernan and Braden 2002, Koronkiewicz *et al.* 2006a). For banded birds along the LCR, up to 93% of adult individuals return to previous breeding sites, while over 38% of juveniles disperse from natal sites (Koronkiewicz *et al.* 2006a). Juvenile dispersal is largely within the regional area, although long distance dispersal has occurred, with movements greater that 200 km reported (McKernan and Braden 2001b, Koronkiewicz *et al.* 2006a). These movements and site fidelity suggest that the Virgin River/LCR population may be a sub-population of a greater meta-population (Koronkiewicz *et al.* 2006a).

Nest building usually begins 3-7 days after pair formulation. On average, one egg is laid per day, with a typical clutch size of four eggs laid within 5 days. Egg laying can start as early as late May, but is usually in early to mid-June (Sogge *et al.* 1997a, Sogge *et al.* 1997b). Willow flycatcher eggs are tan to buffy in color, with a few brown spots on one end. They are approximately 18 mm long and 14 mm wide (Sogge *et al.* 1997a). Upon completion of egg laying, the female usually incubates the eggs for approximately 12 days, and all eggs usually hatch within 24-48 hours of one another. Nestlings fledge usually within 12-15 days (Paxton and Owen 2002). Chicks are usually present from mid-June through early August. SWFL will re-nest, either after the first nest fledges or after failure, and have been documented to have up to four nesting attempts and three clutches (Sferra *et al.* 1997, McKernan and Braden 2001b, Koronkiewicz *et al.* 2006). Adults depart from breeding territories as early as mid-August, but may stay until mid-September if nesting was late. Fledglings usually leave the breeding areas a week or two after adults (Sogge *et al.* 1997a).

SWFL build open cup nests that are approximately 8 cm high and 8 cm wide with dangling material below. Nests are typically placed within the fork of branches with the nest cup supported by several stems. Nest height varies and can be anywhere from ground height to several meters high, depending on height of nest tree. Typical nest height is about 2 to 7 meters (Sogge *et al.* 1997a). Flycatchers nest in various tree species including Goodding's willow, coyote willow, cottonwood, saltcedar, boxelder, and other native and exotic tree species. Along the LCR, main nest substrates include Goodding's willow (approx. 20-30%), coyote willow (approx. 5-15%), Fremont cottonwood (approx. 5%), and saltcedar (approx. 50%-70%). In some areas, such as Topock Marsh, nearly 100% of the nests are in saltcedar (McKernan and Braden 2001b, Koronkiewicz *et al.* 2004, 2006a, McLeod 2005).

Nest success averages from 40 to 50% through all years of study along the lower Colorado River (McKernan and Braden 1997, 1998, 1999, 2001a, 2001b, 2002, 2006, Koronkiewicz *et al.* 2004, 2006a, McLeod 2005) and approximately 25-70% over the complete range of the SWFL (USFWS 2002). Predation has been the leading cause of nest failure at many study sites throughout the range (USFWS 2002), including along the LCR (McKernan and Braden 2001b and 2002, Koronkiewicz *et al.* 2004, 2006a, McLeod 2005). Predation has averaged 33-65% along the LCR from 1996 through 2005 (McKernan and Braden 2001b and 2002, Koronkiewicz *et al.* 2004, 2006a, McLeod 2005). For Arizona statewide surveys in 2005, approximately 77% of failed nests were due to depredation (English *et al.* 2006). Although these numbers are within the typical range for open-cup nesting passerine birds (USFWS 2002), this amount of predation increases the stress on a species already endangered.

Parasitism by brown-headed cowbirds is another cause of nest failure. Cowbird parasitism may impact some SWFL populations enough to warrant management actions. The cowbird lays it eggs in the nest of the host species, and the host then incubates the cowbird eggs, which typically hatch prior to the hosts own young. In some cases, the cowbird female or even the chick will push the host species eggs out of the nest, thereby acting as nest predators (USFWS 2002, Koronkiewicz *et al.* 2006a). Parasitism rates along the LCR have ranged from 0 to 75% in some areas, with the average parasitism rate in 2005 at 32% for all sites (Koronkiewicz *et al.* 2006a). The Arizona statewide average for 2005 was 7% (English *et al.* 2006). The effects of brood parasitism may reduce the likelihood that a SWFL nest containing eggs will fledge young. Ongoing studies are being conducted on the effectiveness of brown-headed cowbird trapping within the life history sites along the LCR and its tributaries (Koronkiewicz *et al.* 2006a).

Abandonment and desertion, although typically low, are also causes of nest failure. Typically, causes for these are unknown, but addled or unfertilized eggs, disturbance, and in some cases brood parasitism may all contribute. Abandonment and desertion accounted for 13% and 9%, respectively, for nest failures at life history study sites along the LCR and tributaries in 2005 (Koronkiewicz *et al.* 2006a). For Arizona statewide surveys, abandonment and desertion together accounted for less than 6% of the nest failures.

Diet

The SWFL is an insectivore that hawks insects while in flight, gleans insects from foliage, and occasionally captures them from the ground (USFWS 2002). Flycatchers forage from within the habitat or above the canopy, above water, or glean from trees and herbaceous cover (McCabe 1991, Sogge 2000, USFWS 2002). The main diet of the flycatcher consists of small to medium size insects such as true bugs (order Hemiptera), wasps and bees (Hymenoptera), flies (Diptera), beetles (Coleoptera), butterflies and caterpillars (Lepidoptera), and spiders (Araneae) (Drost *et al.* 1998, McCabe 1991, Sogge 2000, Drost *et al.* 2001, DeLay *et al.* 2002, Durst 2004). Berries and small fruits have also been reported but are typically rare (McCabe 1991). The flycatcher can exploit a diverse array of insects depending on availability within the habitat (Drost *et al.* 1998, Drost *et al.* 2001, Drost *et al.* 2003, DeLay *et al.* 2002, Durst 2004). Diet may differ between sites and between years depending on abundance and availability of insects in and near the breeding habitat (DeLay *et al.* 2002, Drost *et al.* 2003, Durst 2004). Although there were differences in prey types consumed by the flycatcher among different habitats (e.g. native versus nonnative), there is no significant differences in the abundance of insects available between habitats (Durst 2004), and there is no evidence that the physiological condition of flycatchers is lower in saltcedar habitats (Owen *et al.* 2005).

Habitat

The SWFL breeds in dense riparian vegetation near surface water or saturated soil, across a large elevational and geographic area (USFWS 2002, Sogge *et al.* 1997a). Dominant plant species consist of large riparian trees such as coyote willow (*Salix exigua*), Goodding's willow (*Salix goodingii*), Fremont cottonwood (*Populus fremontii*), boxelder (*Acer negundo*), and nonnative saltcedar (*Tamarix* spp.) and Russian olive (*Elaeagnus angustifolia*) (USFWS 2002). Habitat can be described in four general types (Sogge *et al.* 1997a, Sogge and Marshall 2000, USFWS 2002):

- Native (>90%)—This habitat type usually consist of dense monotypic stands of native vegetation such as cottonwood and willow, ranging from 3 to 7 m in height, often associated with sedges, rushes, and other wetland plants. Structure is usually very dense at the 2-m level. Water or moist soil is typically present.
- Mixed native/exotic (>50% native)—This habitat type consists of greater that 50% natives such as cottonwood and willow, intermixed with saltcedar or Russian olive. Structurally, the habitat has multiple layers with a canopy height of 4-10 m. The lower 2 m is usually hard to penetrate due to the density. Again, water or moist soil is typically present.
- Mixed exotic/native (>50% exotic)—This habitat type is similar to the mixed native/exotic but consists of greater that 50% exotic. Exotics make up primarily the understory, with the overstory typically being large, sparse cottonwood or Goodding's willow, and water or moist soil is typically present.
- Exotic (>90%)—This habitat type is dominated by nonnative exotic vegetation such as saltcedar or Russian olive. Canopy height averages 5 to 10 m, and is usually dense and closed. The lower 2 m is usually dead or dying branches, with little to no understory present. Water or, at a minimum, moist soil is present.

Occupied sites vary in size and shape but all are relatively dense, with some open areas, and are usually associated with open or standing water. Occupied patches can be as small as 0.8 ha and as large as several hundred hectares, but are typically greater than 10 m wide. Although most of the sites are associated with open water, marshy seeps, or saturated soil where the nest tree can be in standing water, hydrologic conditions can change drastically during the breeding season and between years (Sogge *et al.* 1997a, Sogge and Marshall 2000, USFWS 2002, Koronkiewicz *et al.* 2006a). Because birds are exposed to extreme environmental conditions throughout the desert Southwest, dense vegetation and moist soils at the nest may be needed to provide a more suitable microclimate for raising young by increasing humidity within the utilized site (Allison *et al.* 2003, Sogge and Marshall 2000, Koronkiewicz *et al.* 2006a).

Habitat models have been developed for habitat selection and distribution in areas such as Roosevelt Lake in Arizona (Hatten and Paradzick 2003, Paradzick 2005, Brodhead 2005). One model used vegetation density, edge habitat, and proximity to patch boundaries, along with width of floodplain and floodplain features, to develop a set of predictor (GIS) variables to map potential SWFL habitat on a large regional scale (Hatten and Paradzick 2003). This model was highly predictive of occupied SWFL habitat in Arizona. A similar model could be used to map potential SWFL habitat along the LCR. However, this model did not use landscape or patch-scale vegetation coupled with hydrologic conditions under the stand (Paradzick 2005). Without the hydrologic data, the predicted area could be overstated, such as areas of upland saltcedar that are not SWFL habitat. Data collected through research and monitoring along the LCR may be used to construct a predictive model for breeding habitat selection in the LCR MSCP planning area.

Vegetation analysis has occurred at life history sites along the LCR and its tributaries since 1996 (McKernan and Braden 2002, Koronkiewicz *et al.* 2006a). Data gathered includes average canopy height, total canopy closure, woody ground cover, distance to nearest standing water or saturated soil, and additional foliage density measurements. Measurements have been taken at the nest, within territory, and at non-use plots. Analysis of this data is still being conducted and will be presented in a final report in 2008. Preliminary analysis suggests that, overall, flycatchers breed in a wide variety of habitats throughout the Virgin River and LCR. These areas contain relatively homogenous, contiguous stands of riparian vegetation that differ from each other both structurally and compositionally. Preliminary nest productivity, as related to vegetation type (e.g., nonnative versus native), shows no significant difference (McKernan and Braden 2002), but further analysis will be conducted.

At all study sites along the LCR and tributaries, habitats usually have high canopy closure, with no distinct understory, overstory, or structural layers (Koronkiewicz *et al.* 2006a). High vegetation volume may be more important than specific tree species type or habitat structure. High vegetation volume and high foliage density at nest sites and within breeding patches has been reported, not only along the LCR (McKernan and Braden 2002, Koronkiewicz *et al.* 2006a), but also in other willow flycatcher breeding areas (Sedgwick and Knopf 1992, Sogge and Marshall 2000, Allison *et al.* 2003, Stoleson and Finch 2003, Paradzick 2005). This factor, along with the presence of water, was consistent throughout the range.

The presence of water is an important component of SWFL habitat (Sogge and Marshall 2000, USFWS 2002). Studies indicate that SWFL nest sites are usually closer to water than non-use sites (Stoleson and Finch 2003, Paradzick 2005, Koronkiewicz *et al.* 2006a). Nest sites are usually located within 200 m of open or standing water and usually contain soils that are higher in water content than non-use sites (McKernan and Braden

2002, Stoleson and Finch 2003, Paradzick 2005, Koronkiewicz *et al.* 2006a). Water and/or moist soils help regulate temperature and relative humidity within the stand, produce the right conditions for insect development and survival, and are associated with creating a greater foliage density (USFWS 2002, Paradzick 2005, Koronkiewicz *et al.* 2006a).

Microclimate within the habitat patch may influence nest site placement, reproductive success, habitat growth and density, and food availability (Koronkiewicz *et al.* 2006a). Microclimate studies currently being conducted consist of measuring temperature, relative humidity, vapor pressure, and soil moisture at the nest, within territory, and at non-use sites (Koronkiewicz *et al.* 2006a). Preliminary results indicate that nests were located in areas with fewer temperature extremes, greater soil moisture, higher relative humidity, and cooler diurnal temperatures (Koronkiewicz *et al.* 2006a). These preliminary results appear to indicate that microclimate may limit nesting habitat suitability, territory location, and nest placement (Koronkiewicz *et al.* 2006a).

Habitat characteristics on the wintering grounds for the willow flycatcher are similar to the characteristics of the breeding habitat. Wintering habitats are strongly associated with standing water and/or saturated soils, patches or stringers of riparian species of trees, woody understory, and open areas such as pastures, savannas, or bodies of water with forested edges (Koronkiewicz and Whitfield 1999, Koronkiewicz and Sogge 2000, Lynn and Whitfield 2002, Lynn *et al.* 2003, Nishida and Whitfield 2005, Koronkiewicz *et al.* 2006b). These sites were influenced by seasonal inundation. Overall size, shape, and species composition varied between sites, but habitat structure and presence of water were similar at all sites. Woody vegetation height ranged from 1 to 3 m high and the density ranged from impenetrable thickets to clumpy sparse patches (Lynn *et al.* 2003). Average canopy heights ranged from 6 to 15 m. Wintering sites were found within 400 m of freshwater lakes, marshes, and/or wetlands (Koronkiewicz and Whitfield 1999, Koronkiewicz and Sogge 2000, Lynn and Whitfield 2002, Lynn *et al.* 2003, Nishida and Whitfield 2005, Koronkiewicz *et al.* 2006b).

Threats

Habitat alteration, as well as loss and fragmentation are considered one of the greatest threats to the SWFL (Marshall and Stoleson 2000). Riparian habitats in the Southwest are naturally patchy and subject to periodic disturbance. Factors contributing to habitat loss include water management, such as dams and reservoirs, diversions and groundwater pumping, channelization and bank stabilization, agricultural development, livestock grazing, phreatophyte control, increased recreation, and urbanization. All of these cause loss of habitat, habitat fragmentation, loss of critical water underneath stands, and human disturbance (Marshall and Stoleson 2000).

These factors have affected both local and regional SWFL populations. For example, all of Arizona’s major rivers and tributaries that have known SWFL breeding habitat have suffered extensive dewatering, overallocation of water rights, and loss and fragmentation of riparian habitats (USFWS 2002). Flycatchers that were once abundant, for example, near the confluence of Gila and Colorado rivers, are now rare at best (McKernan and Braden 1999 and 2001b, Koronkiewicz *et al.* 2006a).

Riparian habitat along the LCR alone has drastically changed from a cottonwood-willow dominated habitat, including approximately 89,200 acres of potential willow flycatcher breeding habitat (Bureau of Reclamation 1999b), to over 80,000 acres of saltcedar, with no over-bank flooding to help rejuvenate native riparian stands (Lower Colorado River Multi-Species Conservation Program (LCR MSCP 2004a.) Currently, it is estimated that only approximately 6,200 acres of SWFL habitat remains along the LCR from Lake Mead to the Southerly International Border with Mexico (LCR MSCP 2004a).

Another major cause of habitat loss is the increased threat of fire. Every year, several thousands of acres of riparian habitat are burned due to human intervention or natural causes such as lightning. Of the 6000 acres of riparian habitat burned between 1996 and 2001, over 900 acres of potential, suitable, and occupied SWFL habitat has been lost on the LCR. (Bureau of Reclamation 1997, 1998, 1999a, 2000, 2002).

Although the SWFL now nests in saltcedar, this has some disadvantages. Saltcedar exudes salts and creates soils that are too salty for other native species to propagate, thus reducing diversity in the stand, which may affect prey base for flycatchers. Saltcedar also is much more adapted to fire and reestablishes more readily than native species, thus changing the composition of the stand, and increasing the chance of greater habitat loss and degradation. Deep root systems and extended production and proliferation of seeding from March through October gives saltcedar selective advantage over natives under stressed conditions such as lack of flooding, and may reduce soil moisture and standing water conditions needed for flycatcher habitat (Marshall and Stoleson 2000).

The SWFL has evolved with predation and cowbird parasitism, but increased populations of predators and cowbirds have become a major threat to some local populations. Predation is the leading cause of nest failure in many populations of SWFL (Marshall and Stoleson 2000, USFWS 2002), including those along the LCR and its tributaries (McKernan and Braden 2002, Koronkiewicz *et al.* 2006a). Known and suspected nest predators include snakes, predatory birds such as raptors, corvids, grackles and cowbirds, small mammals, and even ants (Marshall and Stoleson 2000). Cowbird populations have expanded greatly with the expansion of livestock grazing, agriculture, and deforestation (Marshall and Stoleson 2000, Siegle and Ahlers 2004). Although predation and parasitism rates are similar to other open cup nesting birds, this coupled with other factors such as habitat loss puts even more stress on the SWFL population (USFWS 2002, Marshall and Stoleson 2000).

Other threats that have not been studied as thoroughly include parasites, disease, and environmental toxins. Internal and external parasites have been recorded, but the extent of impacts has not been determined. Diseases such as West Nile Virus and Avian Flu are new threats that so far have not gotten into the SWFL population, but could have a devastating effect if they do, due to the small population size and the semi-colonial aspect of the SWFL. Environmental toxins may also play a factor as populations close to agriculture and human habitats, such as golf courses, increase the possibility of toxins entering into the diet of the flycatcher. Although this has not been studied to any great extent, bill deformities and missing eyes have been reported from birds at sites in Arizona, Colorado, and New Mexico. A study was completed on environmental contaminants in surrogate birds and insects found in SWFL habitat in Arizona. This study showed an accumulation of inorganic elements in eggshells and contents of eggs, although the only contaminant in this study with unusually high levels was strontium (Mora 2002). Increased concentrations of this metal may be associated with decreased egg production, and potentially higher embryonic mortality, but further studies are needed.

LCR MSCP Conservation Measures

WIFL1—Create 4,050 acres of southwestern willow flycatcher habitat (LCR MSCP 2004a). Of the 5,940 acres of created cottonwood-willow, at least 4,050 acres will be designed and created to provide habitat for this species. Created cottonwood-willow will be designed and managed to support cottonwood-willow types I–IV that provide breeding habitat for this species. The created cottonwood-willow would also function as migration habitat for birds that migrate along the LCR. A total of 2,700 acres of created habitat will be designed and managed to provide habitat for both the southwestern willow flycatcher and yellow-billed cuckoo. To provide habitat for both species, created habitat will need to be composed of cottonwood-willow types I–IV, include moist soils for flying insect production, and be in large habitat blocks (at least 25 acres but preferably up to 200 or more acres). The remaining 1,350 acres of the 4,050 acres of created habitat will also be composed of cottonwood-willow types I–IV and will include moist soils, but patches of this habitat may be smaller if site constraints limit the construction of larger habitat patches.

Of the 1,350 acres of habitat to be created specifically for the yellow-billed cuckoo (Section 5.7.14), patches that provide surface water or moist surface soil conditions during the breeding season will also support habitat for the southwestern willow flycatcher.

In addition to the spatial replacement of affected habitat, the quality of created habitat will be substantially greater than the affected habitat. Affected southwestern willow flycatcher habitat is dominated by dense stands of saltcedar that support little vegetative diversity relative to the cottonwood-willow land cover that will be created and managed as flycatcher breeding habitat. Cottonwood-willow land cover created to provide southwestern willow flycatcher habitat will be designed and managed to be dominated by native riparian trees (i.e., cottonwood and willow trees), support flying insect production used as food by the flycatcher, support a diversity of plant species, provide a dense multilayered canopy, support multiple seral stages, and provide substantial areas of edge habitat. Created habitat, thus, will be similar to the condition of the species’ native habitat that was historically present along the LCR.

The relative suitability and carrying capacity of saltcedar and cottonwood-willow habitats for nesting southwestern willow flycatchers are difficult to measure under current conditions because saltcedar now dominates most riparian areas along the LCR. Based on historical accounts, however, cottonwood-willow forests of the LCR once supported a high diversity and density of nesting birds, including willow flycatchers (Grinnell 1914, Garrett and Dunn 1981, Rosenberg *et al.* 1991). Thus, it is reasonable to assume that the successful replacement of the current saltcedar-dominated habitats by the species' historical, native habitat would provide highly favorable conditions for long-term maintenance and enhancement of southwestern willow flycatcher populations on the LCR.

To ensure that high quality and fully functioning southwestern willow flycatcher breeding habitat is created, the following design and management criteria, subject to adjustment through the LCR MSCP adaptive management process, will be applied to created cottonwood-willow land cover dedicated as replacement southwestern willow flycatcher habitat:

- Southwestern willow flycatcher habitat will be created in patches of at least 10 acres, with an objective of creating larger patches of habitat.
- Created-habitat patches will be close to each other or existing tracts of riparian forest that provide southwestern willow flycatcher habitat in a manner that will maximize continuity with other riparian habitats.
- Designs of created habitats will emphasize creation of nesting habitat within 200 feet of standing or slow-moving water or moist surface soils (suitable insect-productive foraging habitats) and will include creation of suitable habitat edges that are preferred by this species.
- Created habitat will include provisions for supporting moist surface soils and standing or slow-moving water required by the species within their territories during the breeding season (may extend from late April to August along the LCR). Maintaining these conditions could involve creation of canals and shallow swales that permanently or seasonally maintain surface water or moist surface soil conditions. Because the actual period that moist soils or ponded or slow-moving water conditions must be present to support successful reproduction is not well understood, watering of created habitat will be managed adaptively to determine periods when water must be present to support flycatcher reproduction.
- Canals and shallow swales may be needed to dissect blocks of created cottonwood-willow that will be wide enough (estimated to be at least 25 feet) to offer the interior forest-edge conditions necessary to support southwestern willow flycatcher habitat, and the microrelief and soil moisture conditions necessary to support a diversity of understory plant species, and also supply irrigation water.

- Created habitat will be designed and actively managed to support a vigorous plant community that will support multiple layers, seral stages, and age cohorts of trees.
- Mounds and depressions, to the extent necessary, will be created in habitat created on conservation areas to establish some topographic diversity that will also provide habitat diversity by increasing plant and insect prey species diversity.

WIFL2—Maintain existing important habitat areas (LCR MSCP 2004a).
The Applicants, under agreements with cooperating land management agencies, will provide funding to those agencies to maintain a portion of existing southwestern willow flycatcher habitat within the LCR MSCP planning area (Section 5.4.2). Maintaining important existing habitat areas is necessary to ensure the continued existence of the southwestern willow flycatcher in the LCR MSCP planning area, provide for the production of individuals that could disperse to and nest in LCR MSCP-created habitats, and support future recovery of the species.

LCR MSCP Research and Monitoring Needs

Currently Reclamation is utilizing a 10-survey protocol for presence/absence studies along the LCR. This protocol should be evaluated for its effectiveness in identifying the actual absence of flycatchers, and determining the dynamics of northbound migration along the LCR (Koronkiewicz *et al.* 2006). SWFL surveys along the LCR have utilized a 10-visit survey protocol—twice the intensity of comparable survey efforts and recommended USFWS protocol—but this protocol may be useful in determining the timing of migrations and identifying areas used during migration (Koronkiewicz *et al.* 2006). The increased time may also lead to identification of banded SWFLs during the migration, although in the past this has been extremely rare; only one confirmed sighting has occurred along the LCR since banding began (Koronkiewicz *et al.* 2006). The current protocol will be evaluated to determine whether added benefits, such as collection of migration data or increased identification of banded individuals, warrant this level of survey effort.

Habitat use by unpaired individuals is largely unknown (Koronkiewicz *et al.* 2006). Studies incorporating telemetry and/or netting in surrounding habitats near known breeding areas may provide data on the use and importance of other habitats. These data may help guide habitat creation efforts by determining whether species such as mesquite and those found in marsh habitat should be included in the overall habitat mosaic. Also, habitat creation sites located within contiguous riparian areas may attract floater or dispersing populations rather than isolated sites (Koronkiewicz *et al.* 2006).

Depredation has been a major cause of SWFL nest failure along the LCR and its tributaries since studies began in 1996. Depredation may be linked to landscape characteristics and fluctuations in predator densities, abundance, and richness. These factors ultimately affect flycatcher nest success, but are complex and not well defined

(Koronkiewicz *et al.* 2006a). A study identifying nest predator assemblages and their habitats may help guide restoration efforts to avoid increasing predator rates. Studies specifically designed to address open-cup nest predators would be helpful not only for the SWFL but for other LCR MSCP covered species such as the yellow-billed cuckoo, Arizona Bell's vireo, Sonoran yellow warbler, and summer tanager.

Cowbird control studies have been inconclusive along mainstem river systems, such as the LCR. These studies suggest that trapping may not increase nest success, especially along larger river corridors (Koronkiewicz *et al.* 2006, Moore 2006, Ryan and White 2006). There may be other means of decreasing parasitism rates through specific planting techniques, establishing other vegetation around the perimeter of habitat, and isolating newly created habitat away from agriculture and urban areas.

Data has been gathered on microclimate, vegetation, and nest sites along the LCR since 1996. These data need to be synthesized into a habitat model that can help determine the parameters needed for restoration of flycatcher habitat. For example, it is known that density and soil moisture are significant components of flycatcher habitat, but the data have yet to be synthesized as to the thresholds for density and soil moisture to create the appropriate microclimate for breeding habitat. It is then imperative to take that information and determine the necessary planting densities and watering regime to create those parameters.

Past habitat creation sites along the lower Colorado River have not resulted in the successful establishment of a breeding population of SWFL. An in-depth analysis of existing habitat creation sites is needed to determine what necessary habitat components to incorporate into future habitat creation. Management techniques must be developed to allow for long-term establishment and management of SWFL breeding habitat, including when and how often to cut trees to retain an earlier successional state, how often and how much water is needed to not only draw SWFL in, but to maintain proper microclimate within the habitat (e.g., soil moisture and/or amount of standing water needed, if any), seasonal timing of irrigation, and tree species mosaic within a stand.

Monitoring of microclimate, vegetation, and ground water conditions between Parker and Imperial dams was initiated in 2005 to address how hydrological changes from water transfers associated with Interim Surplus Criteria and Secretarial Implementation Agreements may affect riparian habitats along the LCR (USFWS 2001). This monitoring program will continue for up to 5 years after implementation of all water transfer actions, unless it becomes part of a broader effort associated with recovery actions, and will be reviewed every 5 years to determine whether this level of effort is appropriate (USFWS 2001). This monitoring effort may help answer questions such as timing of SWFL migration, whether microclimatic conditions in habitats below Parker Dam are limiting for breeding flycatchers, and what effects lowering river levels have on current habitat.

Other Potential Research and Monitoring Opportunities

The Southwestern Willow Flycatcher Final Recovery Plan was completed in 2002 (USFWS 2002). Range-wide research and monitoring opportunities are described within this document.

Literature Cited

Allison, L. J., C. E. Paradzick, J. W. Rourke, and T. D. McCarthey. 2003. A characterization of vegetation in nesting and non-nesting plots for southwestern willow flycatchers in central Arizona. Studies in Avian Biology 26:81-90.

AOU. 1973. 32nd checklist supplement. Auk 90 (2):415-416.

Bakian, A. V. and E. H. Paxton. 2004. Home range, movement, and habitat use of willow flycatchers, Fish Creek, Utah—2003. U.S. Geological Survey Report to the Bureau of Reclamation, Salt Lake City, UT. 24 p.

Braden, G. T., and R. L. McKernan. 1998. Nest cycles, vocalizations, and survey protocols of the endangered southwestern willow flycatcher (*Empidonax traillii extimus*). Report to Bureau of Reclamation, Lower Colorado Region, Boulder City, NV. 36 p.

Brodhead, K. M. 2005. The influence of riparian-canopy structure and coverage on the breeding distribution of the southwestern willow flycatcher. M.S. Thesis. Montana State University, Bozeman, MT. 112 p.

Browning, M. R. 1993. Comments on the taxonomy of *Empidonax traillii* (willow flycatcher). Western Birds 24:241-257.

Bureau of Reclamation. 1997. Fire occurrence along the lower Colorado River in potential or occupied southwestern willow flycatcher habitat from October 1996 through July 1997. Bureau of Reclamation, Lower Colorado Region, Boulder City, NV. 8 p. + Appendices.

Bureau of Reclamation. 1998. Fire occurrence along the lower Colorado River in potential or occupied southwestern willow flycatcher habitat from August 1997 through August 1998. Bureau of Reclamation, Lower Colorado Region, Boulder City, NV. 9 p. + Appendices.

Bureau of Reclamation. 1999a. Fire occurrence along the lower Colorado River in potential or occupied southwestern willow flycatcher habitat from August 1998 through August 1999. Bureau of Reclamation, Lower Colorado Region, Boulder City, NV. 8 p. + Appendices.

Bureau of Reclamation. 1999b. Long-term restoration program for the historical southwestern willow flycatcher (*Empidonax traillii extimus*) habitat along the lower Colorado River. Report submitted by Bureau of Reclamation, Lower Colorado Region to Lower Colorado River Multi-Species Conservation Program, Boulder City, NV. 70 p.

Bureau of Reclamation. 2000. Fire occurrence along the lower Colorado River in potential or occupied southwestern willow flycatcher habitat from August 1999 through August 2000. Bureau of Reclamation, Lower Colorado Region, Boulder City, NV. 10 p. + Appendices.

Bureau of Reclamation. 2002. Fire occurrence along the lower Colorado River in potential or occupied southwestern willow flycatcher habitat from September 2000 through August 2001. Bureau of Reclamation, Lower Colorado Region, Boulder City, NV. 8 p. + Maps.

Cardinal, S. N., and E. H. Paxton. 2005. Home range, movement, and habitat use of the southwestern willow flycatcher, Roosevelt Lake, AZ—2004. U.S. Geological Survey report to Bureau of Reclamation, Phoenix. 26 p.

Cowardin, L. M., V. Carter, F. C. Golet, and E. T. LaRoe. 1979. Classification of wetlands and deepwater habitats of the United States. U.S. Department of the Interior, Fish and Wildlife Service, Washington, D.C. Jamestown, ND: Northern Prairie Wildlife Research Center Home Page. http://www.npwrc.usgs.gov/resource/1998/classwet/classwet.htm (Version 04DEC98).

DeLay, L. S., S. H. Stoleson, and M. Farnsworth. 2002. A quantitative analysis of the diet of southwestern willow flycatchers in the Gila Valley, New Mexico. Final Report to T&E, Inc. Cortaro, AZ. 13 p.

Drost, C. A., M. K. Sogge, and E. Paxton. 1998. Preliminary diet study of the endangered southwestern willow flycatcher. USGS Colorado Plateau Research Station, Flagstaff, AZ. Report to Bureau of Reclamation, Phoenix, AZ.

Drost, C. A., E. H. Paxton, M. K. Sogge, and M. J. Whitfield. 2001. Food habitats of the endangered southwestern willow flycatcher. Final report to Bureau of Reclamation, Salt Lake City. USGS Forest and Rangeland Ecosystem Science Center, Colorado Plateau Field Station, Flagstaff, AZ. 24 p.

Drost, C. A., E. H. Paxton, M. K. Sogge, and M. J. Whitfield. 2003. Food habitats of the southwestern willow flycatcher during the nesting season. Studies in Avian Biology 26:96-103.

Durst, S. L. 2004. Southwestern willow flycatcher potential prey base and diet in native and exotic habitats. M.S. Thesis. Northern Arizona University, Flagstaff, AZ. 86 p.

Durst, S. L., M. K. Sogge, H. C. English, S. O. Williams III, B. E. Kus, and S. J. Sferra. 2005. Southwestern willow flycatcher breeding site and territory summary—2004. USGS Southwest Biological Science Center, Colorado Plateau Research Station report to Bureau of Reclamation. 18 p.

English, H. C., A. E. Graber, S. D. Stump, H. E. Telle, and L. A. Ellis. 2006. Southwestern willow flycatcher 2005 survey and nest monitoring report. Nongame and Endangered Wildlife Program Technical Report 248. Arizona Game and Fish Department, Phoenix, AZ.

Garrett, K., and J. Dunn. 1981. Birds of southern California: status and distribution. The Artisan Press, Los Angeles, CA.

Grinnell, J. 1914. An account of the mammals and birds of the Lower Colorado Valley, with especial reference to the distributional problems present. University of California Publications in Zoology 12(4):51-294 as found in reprint from Arno Press, New York 1978. 294 p.

Hatten, J. R. and C. E. Paradzick. 2003. A multiscaled model of southwestern willow flycatcher breeding habitat. Journal of Wildlife Management 67(4):774-788.

Koronkiewicz, T. J., and M. J. Whitfield. 1999. Surveys for wintering willow flycatchers (*Empidonax traillii*) in Costa Rica and Panama. Final report submitted to Bureau of Reclamation, Phoenix, AZ. 91 p.

Koronkiewicz, T. J., and M. K. Sogge. 2000. Willow flycatcher (*Empidonax traillii*) winter ecology in Costa Rica: 1999/2000. USGS Forest and Rangeland Ecosystem Science Center, Colorado Plateau Field Station, Flagstaff, AZ. Submitted to Bureau of Reclamation, Phoenix, AZ (Intra-agency Agreement No. 99-AA-32-002). 28 p.

Koronkiewicz, T. J., M. A. McLeod, B. T. Brown, and S. W. Carothers. 2004. Southwestern willow flycatcher surveys, demography, and ecology along the lower Colorado River and tributaries, 2003. Annual report submitted to Bureau of Reclamation, Boulder City, NV, by SWCA Environmental Consultants, Flagstaff, AZ. 125 p.

Koronkiewicz, T. J., M. A. McLeod, B. T. Brown, and S. W. Carothers. 2006a. Southwestern willow flycatcher surveys, demography, and ecology along the lower Colorado River and tributaries, 2005. Annual report submitted to Bureau of Reclamation, Boulder City, NV, by SWCA Environmental Consultants, Flagstaff, AZ. 176 p.

Koronkiewicz, T. J., M. K. Sogge, C. Van Riper III, and E. H. Paxton. 2006b. Territoriality, site fidelity, and survivorship of willow flycatchers wintering in Costa Rica. Condor 108:558-570.

Lower Colorado River Multi-Species Conservation Program. 2004a. Lower Colorado River Multi-Species Conservation Program, Volume II: Habitat Conservation Plan. Final. December 17. (J&S 00450.00) Sacramento, CA.

Lower Colorado River Multi-Species Conservation Program. 2004b. Lower Colorado River Multi-Species Conservation Program, Volume III: Biological Assessment. Final. December 17. (J&S 00450.00.) Sacramento, CA.

Lynn, J. C., and M. J. Whitfield. 2002. Winter distribution of the willow flycatcher (*Empidonax traillii*) in Mexico. Southern Sierra Research Station, Weldon, CA. Submitted to Bureau of Reclamation, Boulder City, NV. 45 p.

Lynn, J. C., T. J. Koronkiewicz, M. J. Whitfield, and M. K. Sogge. 2003. Willow flycatcher winter habitat in El Salvador, Costa Rica, and Panama: characteristics and threats. Studies in Avian Biology 26:41-51.

McCabe, R. A. 1991. The Little Green Bird, Ecology of the Willow Flycatcher. Madison, Wisconsin. 171 p.

McKernan, R. L. 1997. Status, distribution, and habitat affinities of the southwestern willow flycatcher along the lower Colorado River Year 1—1996. Report submitted to Bureau of Reclamation, Lower Colorado River Region, and U.S. Fish and Wildlife Service, Carlsbad Field Office. San Bernardino County Museum, Redlands, CA. 42 p.

McKernan, R. L., and G. Braden. 1998. Status, distribution, and habitat affinities of the southwestern willow flycatcher along the lower Colorado River Year 2—1997. Report submitted to Bureau of Reclamation, Lower Colorado River Region, and U.S. Fish and Wildlife Service, Carlsbad Field Office. San Bernardino County Museum, Redlands, CA. 64 p.

McKernan, R. L., and G. Braden. 1999. Status, distribution, and habitat affinities of the southwestern willow flycatcher along the lower Colorado River Year 3—1998. Report submitted to Bureau of Reclamation, U.S. Fish and Wildlife Service, and Bureau of Land Management. San Bernardino County Museum, Redlands, CA. 71 p.

McKernan, R. L., and G. Braden. 2001a. Status, distribution, and habitat affinities of the southwestern willow flycatcher along the lower Colorado River Year 4—1999. Report submitted to Bureau of Reclamation, U.S. Fish and Wildlife Service, and Bureau of Land Management. San Bernardino County Museum, Redlands, CA. 83 p.

McKernan, R. L., and G. Braden. 2001b. Status, distribution, and habitat affinities of the southwestern willow flycatcher along the lower Colorado River Year 5—2000. Report submitted to Bureau of Reclamation, U.S. Fish and Wildlife Service, and Bureau of Land Management. San Bernardino County Museum, Redlands, CA. 86 p.

McKernan, R. L., and G. Braden. 2002. Status, distribution, and habitat affinities of the southwestern willow flycatcher along the lower Colorado River Year 6—2001. Report submitted to Bureau of Reclamation, U.S. Fish and Wildlife Service, and Bureau of Land Management. San Bernardino County Museum, Redlands, CA. 58 p. + Appendix.

McKernan, R. L., and G. Braden. 2006. Status, distribution, and habitat affinities of the southwestern willow flycatcher along the lower Colorado River Year 7—2002. Final Report—Revised. Report submitted to Bureau of Reclamation and U.S. Fish and Wildlife Service. San Bernardino County Museum, Redlands, CA. 93 p.

McLeod, M. A., T. J. Koronkiewicz, B. T. Brown, and S. W. Carothers. 2005. Southwestern willow flycatcher surveys, demography, and ecology along the lower Colorado River and tributaries, 2004. Annual report submitted to Bureau of Reclamation, Boulder City, NV, by SWCA Environmental Consultants, Flagstaff, AZ. 155 p.

Marshall, R. M., and S. H. Stoleson. 2000. Threats. Chapter 3 *In* D. Finch and S. Stoleson (eds.). Status, ecology, and conservation of the southwestern willow flycatcher. USDA Forest Service, Rocky Mountain Research Station, Albuquerque, NM.

Moore, D. 2005. Status and monitoring of southwestern willow flycatchers within Elephant Butte Reservoir, New Mexico. Bureau of Reclamation, Denver Technical Service Center. Prepared for Albuquerque Area Office, Albuquerque, NM. 34 p.

Moore, D. 2006. Riparian obligate nesting success as related to cowbird abundance and vegetation characteristics along the Middle Rio Grande, New Mexico. Bureau of Reclamation, Technical Service Center, Denver, CO. 22 p. + Appendix.

Mora, M. A. 2002. Environmental contaminants in surrogate birds and insects inhabiting southwestern willow flycatcher habitat in Arizona. Final report submitted to Bureau of Reclamation by USGS Columbia Environmental Research Center, Texas. 43 p.

Nishida, C., and M. J. Whitfield. 2005.Winter distribution of the willow flycatcher (*Empidonax traillii*) in Ecuador and Southern Mexico: 2004. Southern Sierra Research Station, Weldon, CA. Submitted to Bureau of Reclamation, Boulder City, NV. 70 p.

Owen, J. C., M. K. Sogge, and M. D. Kern. Habitat and sex differences in physiological condition of breeding southwestern willow flycatchers (*Empidonax traillii extimus*). The Auk 122(4):1261-1270.

Paradzick, C. E. 2005. Southwestern willow flycatcher habitat selection along the Gila and Lower San Pedro rivers, Arizona: vegetation and hydrogeomorphic considerations. M.S. Thesis. Arizona State University, AZ. 148 p.

Paxton, E. H. 2000. Molecular genetic structuring and demographic history of the willow flycatcher (*Empidonax traillii*). M.S. Thesis. Northern Arizona University, Flagstaff, AZ. 33 p.

Paxton E. H., and J. C. Owen. 2002. An aging guide for willow flycatcher nestlings. Colorado Plateau Field Station, Northern Arizona University, Flagstaff, AZ. 18 p.

Pearson, T. 2002. Polygyny and extra-pair paternity in a population of southwestern willow flycatchers (*Empidonax traillii extimus*). M.S. Thesis. Northern Arizona University, Flagstaff, AZ. 33 p.

Phillips, A. R. 1948. Geographic variation in *Empidonax traillii*. Auk 65:507-514.

Phillips, A. R., J. Marshall, and G. Monsoon. 1964. The Birds of Arizona. University of Arizona Press, Tucson, AZ. 212 p.

Rosenberg, K. V., R. D. Ohmart, W. C. Hunter, and B. W. Anderson. 1991. The Birds of the Lower Colorado River. University of Arizona Press, Tucson, AZ.

Ryan, S., and L. White. 2006. Brown-headed cowbird control program, results of follow-up monitoring—years 2002-2005, Alamo Lake State Wildlife Area and Bill Williams National Wildlife Refuge, Arizona. Bureau of Reclamation, Technical Service Center, Denver, CO. 37 p. + Appendix.

Sedgwick, J. A. 2001. Geographic variation in the song of willow flycatchers: differentiation between *Empidonax traillii adastus* and *E. t. extimus*. The Auk 118(2):366-379.

Sedgwick, J. A., and F. L. Knopf. 1992. Describing willow flycatcher habitats: scale perspectives and gender differences. Condor 94:720-733.

Sferra, S. J., T. E. Corman, C. E. Paradzick, J. W. Rourke, J. A. Spencer, and M. W. Sumner. 1997. Arizona Partners In Flight southwestern willow flycatcher survey: 1993-1996 summary report. Nongame and Endangered Wildlife Program Technical Report 113. Arizona Game and Fish Department, Phoenix, AZ.

Siegle, R., and D. Ahlers. 2004. Brown-headed Cowbird Management Techniques Manual. U.S. Department of the Interior, Bureau of Reclamation, Technical Service Center, Denver, CO. 58 p. + Appendices.

Sogge, M. K. 1995. Southwestern willow flycatcher (*Empidonax traillii extimus*) monitoring at Tuzigoot National Monument. 1995 progress report to the National Park Service. National Biological Service, Colorado Plateau Research Station/Northern Arizona University. 26 p.

Sogge, M. K., R. M. Marshall, S. J. Sferra, and T. J. Tibbitts. 1997a. A southwestern willow flycatcher natural history summary and survey protocol. National Park Service Technical Report NPS/NAUCPRS/NRTR-97/12. 37 p.

Sogge, M. K., T. J. Tibbitts, and J. R. Petterson. 1997b. Status and breeding ecology of the southwestern willow flycatcher in the Grand Canyon. Western Birds 28:142-157.

Sogge, M. K. 2000. Breeding season ecology. Chapter 6 *In* D. Finch and S. Stoleson (eds.). Status, ecology, and conservation of the southwestern willow flycatcher. USDA Forest Service, Rocky Mountain Research Station, Albuquerque, NM.

Sogge, M. K., and R. M. Marshall. 2000. A survey of current breeding habitats. Chapter 9 *In* D. Finch and S. Stoleson (eds.). Status, ecology, and conservation of the southwestern willow flycatcher. USDA Forest Service, Rocky Mountain Research Station, Albuquerque, NM.

Stoleson, S. H., and D. M. Finch. 2003. Microhabitat use by breeding southwestern willow flycatchers on the Gila River, New Mexico. Studies in Avian Biology 26:91-95.

Unitt, P. 1987. *Empidonax traillii extimus*: an endangered subspecies. Western Birds 18 (3):137-162.

U.S. Fish and Wildlife Service. 1995. Final rule determining endangered status for the southwestern willow flycatcher (*Empidonax traillii extimus*). Federal Register 60:10694 (February 27, 1995).

U.S. Fish and Wildlife Service. 2000. Southwestern Willow Flycatcher Protocol Revision 2000. Albuquerque, NM.

U.S. Fish and Wildlife Service. 2001. Biological Opinion for Interim Surplus Criteria, Secretarial Implementation Agreements, and Conservation Measures on the Lower Colorado River, Lake Mead to the Southerly International Boundary, Arizona, California, and Nevada. Final Biological Opinion, U.S. Fish and Wildlife Service, Phoenix, AZ. 90 p.

U.S. Fish and Wildlife Service. 2002. Southwestern Willow Flycatcher Recovery Plan. Albuquerque, New Mexico. i-ix + 210 p., Appendices A-O.

U.S. Fish and Wildlife Service. 2005. Endangered and Threatened Wildlife and Plants; Designation of Critical Habitat for the Southwestern Willow Flycatcher (*Empidonax traillii extimus*); Final Rule. 50 CFR Part 17. Federal Register 70:60886-61009 (October 19, 2005).

Yard, H. K., and B. T. Brown. 2003. Singing behavior of southwestern willow flycatchers in Arizona. Studies in Avian Biology 26:125-130.

Yong, W., and D. M. Finch. 1997. Migration of the willow flycatcher along the middle Rio Grande. Wilson Bulletin 109:253-268.

DESERT TORTOISE MOHAVE POPULATION
(*Gopherus agassizii*)

Legal Status

The Mohave population of the desert tortoise (*Gopherus agassizii)* was listed as threatened on April 2, 1990. Critical habitat was designated on February 8, 1994, in portions of the Mohave and Colorado Deserts totaling 6.4 million acres (USFWS 1994). A recovery plan for the desert tortoise (Mohave population) was published in 1994 by the Desert Tortoise Recovery Team headed by the United States Fish and Wildlife Service (USFWS 1994). A Desert Tortoise Recovery Plan Assessment was published in 2004 by the Desert Tortoise Recovery Plan Assessment Team which was assembled by the USFWS and headed by the University of Reno (Tracy *et al.* 2004). The Recovery Plan Assessment is a review and assessment of new research and information gathered on many aspects of desert tortoise ecology, threats, conservation biology, monitoring and recovery actions since 1994. The 1994 recovery plan identified five criteria that must be completed before delisting of the tortoise: 1) as determined by a scientifically credible monitoring plan, the population within a recovery unit must exhibit a statistically significant upward trend or remain stationary for at least 25 years (one desert tortoise generation); 2) enough habitat must be protected within a recovery unit, or the habitat and desert tortoise populations must be managed intensively enough, to ensure long-term viability; 3) provisions must be made for population management within each recovery unit so that discrete population growth rates (lambdas) are maintained at or above 1.0; 4) regulatory mechanisms or land management commitments must be implemented that provide for long-term protection of desert tortoises and their habitat; and 5) the population in the recovery unit is unlikely to need protection under the Endangered Species Act in the foreseeable future (USFWS 1994).

The desert tortoise is a species of special concern in the State of Arizona, threatened in the State of California, and a species of conservation priority in the State of Nevada.

Distribution

Historical Range

The desert tortoise was historically distributed in the Mohave and Sonoran Deserts in south central California, southern Nevada, southeastern Arizona, southwestern Utah, and Sonora and northern Sinaloa, Mexico (Stebbins 1954, 1966 *in* USFWS 1994).

Current Range

The desert tortoise has declined throughout its historical range and has been extirpated in parts of its range (Spang *et al.* 1988 *in* USFWS 1994; Berry 1978 *in* USFWS 1994). The desert tortoise is divided into two populations, the Sonoran and Mohave populations, based on genetic and morphological characteristics (Glenn *et al.* 1990 *in* USFWS 1994;

Lamb *et al.* 1989 *in* USFWS 1994; Weinstein and Berry 1987 *in* USFWS 1994). These two distinct populations are recognized under the Endangered Species Act (USFWS in AGFD 2002). The Mohave population occurs north and west of the Colorado River, and the Sonoran population occurs south and east of the Colorado River (USFWS *in* AGFD 2002; USFWS *in* AGFD 2002)

The Mohave population of the desert tortoise inhabits parts of the Mohave Desert in Inyo, Kern, Los Angeles, San Bernardino, and Riverside counties in California; northwestern part of Mohave County in Arizona; Clark County and the southern parts of Esmeralda, Nye, and Lincoln Counties, Nevada; and part of Washington County, Utah (USFWS 1994). The population also inhabits the Colorado Desert, a division of the Sonoran Desert, in Imperial County, San Bernardino County, and Riverside County, California (USFWS 1994). The Mohave population range in Arizona extends north and west of the Colorado River; west of the Beaver Dam Mountains; north of the Virgin Mountains; and in the Pakoon Basin in extreme northwest Mohave County (AGFD 2002). Six evolutionary distinct population units have been identified in the Mohave population: 1) northern Colorado Desert; 2) eastern Colorado Desert; 3) upper Virgin River; 4) eastern Mohave Desert; 5) northeastern Mohave Desert; and the 6) western Mohave Desert. These populations segments were based on genetics, morphology, behavior, ecology and habitat use (USFWS 1994). In the 2004 Desert Tortoise Recovery Plan Assessment, the evolutionary distinct units were revised, based on current genetic evidence, to: 1) Upper Virgin River Desert; 2) Lower Virgin River Desert; 3) Northeastern Mohave Desert; 4) East Mohave Desert; 5) Colorado Desert; and 6) Western Mojave Desert (Rainboth *et al.* 1989 *in* Tracy *et al.* 2004; Lamb *et al.* 1989 *in* Tracy *et al.* 2004; Lamb and Lydehard 1994 *in* Tracy *et al.* 2004; Britten *et al.* 1997 *in* Tracy *et al.* 2004; Tracy *et al.* 2004).

Trend analysis, from long-term study plots established in California during the 1970s and in Nevada and Utah during the 1980s, showed that population declines have occurred in the western part of the desert tortoise’s range in the Mohave Region (Berry 1984 *in* Tracy *et al.* 2004). In California’s western Mohave, populations may have declined nearly 90% since 1940, and as much as 70% locally from 1976 to 1984 (Berry 1984 *in* Natureserve 2006). At the Desert Tortoise Natural Area (Kern County, California), the past 10-year decline has reduced the tortoise population by 88%; a similar 84% decline has been reported for Johnson Valley (USFWS 1994 *in* Natureserve 2006). Furthermore, declines have been reported in the area that was the Western Mohave Desert Recovery Unit in the 1994 recovery plan and the area that is the Western Mohave Desert Distinct Population Segment in the 2004 revision of these areas (Tracy *et al.* 2004). The downward trend in desert tortoise density in this area has continued since the 1994 recovery plan was completed. Spatial analysis showed that areas of decline were greatest in portions of the Fremont-Kramer Desert and the northwestern part of the Superior Cronese Desert Wildlife Management Areas (Tracy *et al.* 2004). Recovery actions implemented in the Western Mohave Recovery Unit since 1994 have not resulted in the reversal of this downward trend (Tracy *et al.* 2004).

Federal agencies that have jurisdiction over desert tortoise habitat in the Mohave Region are the Bureau of Land Management (BLM), the National Park Service (NPS), the

Department of Defense (DOD), the Bureau of Indian Affairs (BIA), and the Bureau of Reclamation (BOR). State parks and wildlife departments also manage desert tortoise habitat (USFWS 1994). The BLM is the primary land manager for the desert tortoise habitat (USFWS 1994).

Populations within LCR MSCP Project Boundary

The Mohave population is present in reaches 1-6 of the Lower Colorado River Multi-Species Conservation Program (LCR MSCP) planning area in an estimated 10,660 acres in desert scrub habitat (LCR MSCP HCP 2004).

The Desert Tortoise (Mohave Population) Recovery Plan (USFWS 1994) described 14 Desert Wildlife Management Areas (DWMA). Land ownership and tortoise densities were estimated for each DWMA, including:

1) Chemehuevi DWMA is located in the Northern Colorado Recovery Unit in San Bernardino County, California. Current densities of desert tortoises are 10 to 275 adults per square mile. The BLM owns 67% of the land, state agencies own 6% of the land; and 25% is privately owned.

2) Chuckwalla DWMA is located in the Eastern Colorado Recovery Unit in Riverside and Imperial Counties, California. Current densities of desert tortoises are 5 to 175 adults per square mile. Land is owned by the BLM, military and private entities.

3) Upper Virgin DWMA is located in the Upper Virgin Recovery Unit in Washington County, Utah. Current densities of desert tortoises are estimated at 250 adults per square mile. Land is owned by the BLM, military and private entities.

4) Fenner DWMA is located in the Eastern Mohave Recovery Unit in San Bernardino County, California. Current densities of desert tortoises are 10 to 350 adults per square mile. Land is owned by Federal agencies (67%), state agencies (5%) and private entities (28%).

5) Ivanpah DWMA is located in the Eastern Mohave Recovery Unit in San Bernardino County, California. Current densities of desert tortoises are 5 to 250 adults per square mile. Land is owned by the BLM.

6) Piute-Eldorado DWMA is located in the Eastern Mohave Recovery Unit in Clark County, Nevada. Current densities of desert tortoises are 40 to 90 adults per square mile. Land is owned by the National Park Service (NPS), BLM and private entities.

7) Beaver Dam Slope DWMA is located in the Northeastern Mohave Recovery Unit in Washington County, Utah, and Mohave County, Arizona. Current densities of

desert tortoises are 5 to 56 adults per square mile. Land is owned by the BLM and private entities.

8) Coyote Spring DWMA is located in the Northeastern Mohave Recovery Unit in Lincoln and Clark Counties, Nevada. Current densities of desert tortoises are 0 to 90 adults per square mile.

9) Gold Butte-Pakoon DWMA is located in the Northeastern Mohave Recovery Unit in Mohave County, Arizona and Clark County, Nevada. Current densities of desert tortoises are 5 to 56 adults per square mile. Land is owned by the BLM, NPS and private entities.

10) Mormon Mesa DWMA is located in the Northeastern Mohave Recovery Unit in Clark and Lincoln Counties, Nevada. Current densities of desert tortoises are 41 to 87 subadults and adults per square mile. Land is owned by the BLM, Union Pacific Railroad, and private entities.

11) Fremont-Kramer DWMA is located in the Western Mohave Recovery Unit in Kern and San Bernardino Counties, California. Current densities of desert tortoises are 5 to 100 adults per square mile. Land is owned by BLM, military, private entities, and state lands.

12) Ord-Rodman DWMA is located in the Western Mohave Recovery Unit in San Bernardino County, California. Current densities of desert tortoises are 5 to 150 adults per square mile. Land is owned by Federal government (65%) and private entities (35%).

13) Superior-Cronese DWMA is located in the Western Mohave Recovery Unit in San Bernardino County, California. Current densities of desert tortoises are 20 to 250 adults per square mile. Land is owned by the BLM (63%), Department of Defense (15%), and private entities (22%).

14) Joshua Tree DWMA is located in the Western Mohave Recovery Unit in Riverside and San Bernardino Counties, California. Current densities of desert tortoise are up to 200 adults per square mile. Land is owned by the NPS.

Land Acquisition

As of 2006, the California Desert District of the BLM has completed the purchase of nearly all the former railroad lands in the Mohave Desert within Desert Wildlife Management Areas, critical habitat, and wilderness areas (Borchard 2006). The army has acquired 93,000 acres of former railroads lands, west of Fort Irwin (Borchard 2006). Ongoing acquisitions are achieved through mitigation agreements, primarily with utilities (Borchard 2006).

Life History

General Description

The desert tortoise is characterized by a high domed shell, brown carapace, yellow plastron without a hinge, and a pattern and prominent growth lines on the plastron and carapace (Stebbins 1985 *in* AGFD 2002). They are also characterized by stocky limbs, forelimbs covered with large conical scales, and a short tail (AGFD 2002). The alveolar ridges of the upper jaws form a sharp angle with each other; jaw margins are serrate. The iris is greenish-yellow or yellow with brown near the outer edge, sometimes brown or mottled. Skin is gray, blackish-gray to black, or reddish-tan (Auffenberg and Franz 1978 *in* Grover and DeFalco 1995; Barker 1964 *in* Grover and DeFalco 1995; Bogert 1954 *in* Grover and DeFalco 1995; Brown 1974 *in* Grover and Defalco 1995; Carr 1952 *in* Grover and DeFalco 1995; Coombs 1977 *in* Grover and Defalco 1995; Ditmars 1930, 1933 *in* Grover and DeFalco 1995; Grant 1936 *in* Grover and DeFalco 1995; Jaeger 1957 *in* Grover and DeFalco 1995; MacMahon 1985 *in* Grover and DeFalco 1995; Stebbins 1966, 1985 *in* Grover and DeFalco 1995; True 1882 *in* Grover and DeFalco 1995). The desert tortoise can be distinguished from the other three species in its genus by the following traits: 1) a rounded front head; 2) an interhumeral seam longer than the integular seam; 3) a single triangular auxiliary scale; and 4) the base of the first claw to the fourth claw equal for the forefoot and hindfoot (Brame and Peerson 1969 *in* USFWS 1994; Auffenberg 1976 *in* USFWS 1994; Crumly 1984 *in* USFWS 1994). Carapace length of adults ranges from 20 to 36 cm (Stebbins 1985 *in* AGFD 2002). The desert tortoise reaches its maximum size at 5-10 years of age (Murray and Klug 1996 *in* AGFD 2002).

Males can be distinguished from females by the following traits: 1) an elongate gular shield; 2) chin glands on each size of the lower jaw that are larger in size than female glands; 3) broader thicker tail and thick toenails; 4) larger size; 5) plastron that is more concave, especially in the femoral area; and 6) dermal ossicles on the thigh and hindfoot are more developed (Auffenberg 1976 *in* Grover and DeFalco 1995; AFGD 2002; Natureserve 2006; Germano *in* AGFD 2002; Bramble 1971 *in* Grover and Defalco 1995; Grant 1936 *in* Grover and DeFalco 1995; Woodbury and Hardy 1948 *in* Grover and DeFalco 1995). Sexing individuals that are less than 15 years old and/or less than 200 mm straight carapace length may be difficult by external morphology alone (Natureserve 2006).

Mohave desert tortoises can be distinguished from Sonoran desert tortoises by their more oval-shaped figure and a higher domed carapace (Germano 1993 *in* AGFD 2002). Sonoran desert tortoises have larger scales on the dorsum of the head and more sharply wedge-shaped snouts than Mohave desert tortoises (Natureserve 2006). Mohave desert tortoise hatchlings are lighter in color and have more serrate surfaces to their marginal scutes than the Sonoran desert tortoise hatchlings (Joyner Griffith 1991 *in* Natureserve 2006). The desert tortoise is the only naturally occurring tortoise in the Mohave region; however, escaped or released captive tortoises of other species are occasionally detected in the Mohave Region (USFWS 1994).

Hatchlings are approximately 4.5 to 5.0 cm long and weigh approximately 20.0 to 27.0 g. They are round in shape and are mustard yellow to brown in color. Edges of scutes are typically brown and centers are dull yellow (Coombs 1977 *in* Grover and DeFalco 1995; Grant 1936 *in* Grover and DeFalco 1995; Jaeger 1955 *in* Grover and DeFalco 1995; Luckenbach 1982 *in* Grover and DeFalco 1995; Miller 1932, 1955 *in* Grover and DeFalco 1995).

Juvenile tortoises (20-25 years old) can be aged by counting concentric annual rings radiating outward from the areolar center of each shell scute. The age of adult desert tortoises (>25 years) is indeterminable due to shell wear and shedding of juvenile rings (Germano 1988 *in* Natureserve 2006).

Breeding

Mating in the Mohave population of the desert tortoise begins in March and April and can extend through October (Black 1976 *in* AGFD 2002; Rostal *et al.* 1994 *in* Natureserve 2006; Goodlett *et al.* 1996 *in* AGFD 2002). The first year of reproductive activity for a female occurs at age 12 to 25; however, size appears to be more of a factor than age for determining the first year of reproduction. When females reach a carapace length of 185 mm, they usually breed (Turner *et al.* 1984 *in* USFWS 1994).

Mate selection is determined by male-male dominance hierarchies and by selective female receptivity (Niblick *et al.* 1994 *in* Natureserve 2006; Burge 1994 *in* Natureserve 2006). Male to male encounters, including head bobbing and ramming, establish social hierarchy (Natureserve 2006). Dominant males are characterized by larger size, longer residency at a site, and past social interactions (Natureserve 2006). Male courting behavior includes approach, head bob, trailing, biting, ramming, sniffing, circling, mounting, shell scratch, hops, grunts, head in and out, and copulation (Ruby and Niblick 1994 *in* Natureserve 2006). Female behavior includes accepting the male by pulling her head in the shell and withdrawing limbs or rejecting the male by walking away (Ruby and Niblick 1994 *in* Natureserve 2006).

The desert tortoise lays its eggs from April through mid-July. The desert tortoise can lay up to three clutches per year, with each clutch containing 5 to 9 eggs (Karl 1998 *in* AGFD 2002; Turner *et al.* 1986 *in* AGFD 2002; Wallis *et al.* 1999 *in* AGFD 2002). Number of clutches per year is dependent on rainfall (Karl 1998 *in* AGFD 2002; Turner *et al.* 1986 *in* AGFD 2002; Wallis *et al.* 1999 *in* AGFD 2002). Incubation period ranges from 85 to 125 days (Spotila *et al.* 1994 *in* Natureserve 2006). Temperatures at the nest site during this period need to be above 26°C and below 35°C for eggs to survive. Drier soils at the nest site are preferred. Soil moisture greater than 4% at the nest site makes the range of suitable temperature narrower (Natureserve 2006). Hatching usually requires 48-72 hours (Natureserve 2006). Hatchlings usually emerge in late summer, but some may overwinter in the nest (Averill-Murray *et al., in press* AGFD 2002). Desert tortoise eggs are pale, elliptical to spherical, brittle shelled, and average 30-40 mm and 20-40 g (Natureserve 2006). Eggs are laid in depressions 3 to 4 inches deep. Eggs are often laid in the most superficial 2 feet of the burrow floor, directly next to the burrow opening, or under the shrub adjacent to the burrow (Barrett 1990 *in* Natureserve 2006). Sex determination is dependent on soil temperature during incubation; males are produced when temperatures are below 31.8°C and females are produced when temperatures are above 31.8°C (Natureserve 2006; Boarman 2002a).

Diet

The desert tortoise feeds on a variety of herbaceous vegetation including annual and perennial grasses, flowers and fruits of annual plants, cacti, and perennial shrubs (Berry 1974 *in* USFWS 1994; Luckenback 1982 *in* USFWS 1994). The desert tortoise will occasionally eat insects, which are a good source of lipid and protein (Grant 1936 *in* Natureserve 2006; Brown 1968 *in* Natureserve 2006; Okomoto 1995 *in* Natureserve 2006; Avery, pers. comm. *in* Natureserve 2006). Forage species selected by tortoises in the west Mohave Desert include: *Astragalus didymocarpus, Astragalus layneae, Camissonia boothii, Euphorbia albomarginatus, Lotus humistratus,* and *Mirabilis bigelovii* (Jennings *in* Boarman 2002a). In the east Mohave Desert, tortoises showed a preference for *Camissonia boothii, Cryptantha angustifolia, Malacothrix glabrata, Opuntia basilaris, Rafinesquia neomexicana, Schismus barbata,* and *Stephanomeria exigua* (Avery 1998 *in* Boarman 2002a). An active adult individual requires approximately 21 kg of herbaceous forage per month (USDI 1991 *in* Natureserve 2006). Diet is based on presence and abundance of forage. Diet consists primarily of annuals during the spring and dry grasses and cactuses during the summer (Minnich 1972, 1979, 1982 *in* Natureserve 2006; Oftendal *et al.* 1995 *in* Natureserve 2006). The desert tortoise is able take advantage of years in which resources are abundant to sustain them through years in which resources are lacking (Nagy and Medica 1986 *in* USFWS 1994; Wallis *et al.* 1992 *in* USFWS 1994). Individuals can tolerate large imbalance in water and energy budgets; adults can survive a year without access to water (Nagy and Medica 1986 *in* USFWS 1994). Desert tortoises can switch from water demanding urea to uric acid for waste elimination, when needed (Cloudsley-Thompson 1971 *in* Natureserve 2006; Minnich 1977 *in* Natureserve 2006; Schmidt-Nielsen and Bentley 1969 *in* Natureserve 2006; Nagy and Medica 1986 *in* Natureserve 2006).

Biology

Activity period for the desert tortoise varies by region, sex, and age class. The Mohave population of the desert tortoise is active from approximately March through October (Minnich 1977 *in* AGFD 2002; Nagy and Medica 1986 *in* AGFD 2002; Peterson 1996 *in* AGFD 2002; Nagy *et al.* 1997 *in* AGFD 2002; Behler and King 1979). They hibernate in burrows the remainder of the year where they conserve water and energy (USFWS 1994). Some individuals may aestivate during dry periods in the summer (Natureserve 2006). Between March through October, activity tends to be bimodal; morning activity begins around 0700 h, with individuals retreating to their burrows from 1100 to 1600 h (Berry 1975 *in* Natureserve 2006; Ruby *et al.* 1994 *in* Natureserve 2006). The desert tortoise maintains its body temperature in the range of 25 to 35°C (Zimmerman *et al.* 1994 *in* Natureserve 2006).

This species demonstrates a delayed maturity and long life (MacArthur and Wilson 1967 *in* USFWS 1994). Existing data is consistent with the possibility that desert tortoises have evolved to exist in metapopulations (Hanski 1999 *in* Tracy *et al.* 2004; Levins and Culver

1971 *in* Tracy *et al.* 2004; Levins *et al.* 1984 *in* Tracy *et al.* 2004). Home range size for desert tortoises ranges from 5 to 50 ha, but individuals may move several kilometers over weeks or years (O'Connor *et al.* 1994 *in* Natureserve 2006; Auffenberg and Iverson 1979 *in* Natureserve 2006; Berry 1986 *in* Natureserve 2006; Barrett 1990 *in* Natureserve 2006). Home ranges of adults are usually larger than those of juveniles and home ranges of males are usually twice the size of female home ranges (O'Connor *et al.* 1994 *in* Natureserve 2006). Production of spring annuals correlates negatively with home range size (Esque *et al. in preparation in* Natureserve 2006; USFWS 1994 *in* Natureserve 2006). Tortoises are inactive 98% of their life, in which they are often subterranean (Nagy and Medica 1986 *in* Natureserve 2006).

Desert tortoise scat is dark brown or black, is approximately 45 mm in length and 20 mm in diameter, and weighs approximately 1.95 g. Size of scat indicates size of tortoise. Scats may serve as territorial markers and may cause subordinates to leave the area (Camp 1916 *in* Grover and DeFalco 1995; Johnson *et al.* 1948 *in* Grover and DeFalco; Coombs 1979 *in* Grover and DeFalco 1995; Luckenbach 1982 *in* Grover and DeFalco 1995; Patterson 1971 *in* Grover and DeFalco 1995; Auffenberg and Weaver 1969 *in* Grover and DeFalco 1995).

Birth rate, survivorship, fecundity, and death rate are all factors in the decline, growth, and stability of desert tortoise populations (Tracy *et al.* 2004). Average annual adult survivorship is approximately 98% in healthy populations. Juvenile survivorship is variable, and believed to be low—approximately 2% for healthy populations (Averill-Murray *et al. in press in* AGFD 2002; Wilbur and Morin 1988 *in* USFWS 1994; Turner *et al.* 1987 *in* USFWS 1994; USFWS 1994). Average annual growth of healthy, non-threatened desert tortoise populations range from 0.5% to 1% (USFWS 1994). Most juvenile mortality is believed to occur in the egg and hatchling stages. The desert tortoise working group ran a population viability analysis and found that, if a population is healthy and relatively free from adult predators, very few juveniles need to survive to adulthood to sustain a viable population (USFWS 1994). According to this analysis, a healthy population of *Gopherus agassizii* should have a density of 10 adults per square mile over an area large enough to support 10,000 to 20,000 adults (USFWS 1994). If the population density is less than the 10 individuals per square mile, there is a high probability of demographic stochasticity, social dysfunction, and genetic deterioration (USFWS 1994). Populations should be managed so that adult mortality does not fall below a lambda of 1.0. A lambda equal to 1.0 would mean a population is neither increasing nor decreasing (USFWS 1994).

Population changes observed in desert tortoise populations have typically followed two patterns: downward trends due to persistent demographic changes or stochastic fluctuations from random events after which the population begins immediate recovery (USFWS 1994). Downward trends will result in extirpation of the population. Large healthy populations should be able to withstand stochastic fluctuations (USFWS 1994).

The desert tortoise is sympatric with the antelope ground squirrel (*Ammospermophilus leucurus*), blacktail jackrabbit (*Lepus californicus*), canyon mouse (*Peromyscus crinitus*), desert cottontail (*Sylvilagus auduboni*), desert woodrat (*Neotoma lepida*), house cat (*Felis domesticus*), kangaroo rat (*Dipodomys merriami*), kit fox (*Vulpes macrotis*), pocket mouse (*Perognathus* spp.), spotted skunk (*Spilogale gracilis*), white-footed mouse (*Peromyscus* spp.), burrowing owl (*Athene cunicularia*), Gambel's quail (*Callipepla gambelii*), poorwill (*Phalaenoptilus nuttallii*), greater roadrunner (*Geococcyx californianus*), banded gecko (*Coleonyx variegates*), coachwhip (*Masticophis flagellum*), desert iguana (*Dipsosaurus dorsalis*), desert spiny lizard (*Sceloporus magister*), gopher snake (*Pituophis melanoleucus*), Mohave green rattlesnake (*Crotalus scutulatus*), sidewinder (*Crotalus cerastes*), spotted night snake (*Hypsiglena torquata*), western rattlesnake (*Crotalus viridis*), western whiptail (*Cnemidophorus tigris*), antlion larva (Myrmeleontidae), black widow (*Lactrodectus mactans*), ground beetle (*Tenebrionidae*), roaches (*Orthoptera*), scorpion (*Centruroides* spp.), silverfish (*Thysanura* spp.), tarantula (*Aphonopelmas* spp.), and ticks (*Acarina, Ornithodoros parkeri*) (Natureserve 2006).

Habitat

The Mohave population of the desert tortoise is found in Mohave desert scrub dominated by creosote bush (*Larrea tridentate*), creosote bursage (*Ambrosia dumosa*), shadscale (*Atriplex*), other sclerophyll shrubs, and small cacti (Germano *et al.* 1994 *in* AGFD 2002). They also occur in Joshua tree (*Yucca brevifolia*) forests and occasionally blackbrush (*Acacia rigidula*) habitat (Germano *et al.* 1994 *in* AGFD 2002). Native desert grasses, particularly galleta grass (*Pleuraphis* spp.) and Indian rice grass (*Acthnotherum hymenoides*), are associated with high desert tortoise densities (Natureserve 2006). The most preferable desert tortoise habitat is where there is a high density of shrubs that provide cover and high densities of perennial and annual forbs and grasses (Berry 1975 *in* Grover and DeFalco 1995; Karl 1980 *in* Grover and DeFalco 1995; Luckenbach 1982 *in* Grover and DeFalco 1995; Schwartzmann and Ohmart 1978 *in* Grover and DeFalco 1995). Desert tortoises prefer sandy loam to rocky soils in valleys, bajadas, and hills (Germano *et al.* 1994 *in* AGFD 2002). Their elevation range is from sea level to 1500 m (Luckenbach 1982 *in* AGFD 2002; Collins *et al.* 1983 *in* AGFD 2002). The desert tortoise basic habitat requirements are sufficient, suitable plants for forage and cover, and suitable substrates for burrow and nest sites (USFWS 1994). Burrows can be up to 10 meters deep and are usually directly below vegetation or in caves in washes (Woodbury and Hardy 1948 *in* AGFD 2002; Burge 1978 *in* AGFD 2002; Luckenbach 1982 *in* AGFD 2002). Desert tortoises prefer areas that receive from 100 mm to 300 mm of rainfall annually (Fritts and Jennings 1994 *in* NatureServe 2006). Anderson *et al.* (2000) found that desert tortoises are more likely to be found in areas with southwest exposures and loamy soils and are least likely to be found in areas of stony soils, northern exposures and areas of very low plant cover.

Burrows used in the spring and summer, when tortoises are active, have the following characteristics: 1) usually larger and longer than the tortoise, often extending 1 to 8 feet in length; 2) a mean floor declination of 15 degrees; 3) opening faces north, northwest, or northeast; 4) often under a shrub; and 5) has a single opening (Burge 1978 *in* Natureserve 2006; Woodbury and Hardy 1948 *in* Natureserve 2006). Burrows used in the winter, when desert tortoises hibernate, have the following characteristics: 1) extends up to 30 feet in length; 2) often used by more than one tortoise; 3) opening faces south; 4) often enhanced by chambers and interconnections between dens; and 5) hold air masses with stable, high relative humidity reaching 40% (Woodbury and Hardy 1948 *in* Natureserve 2006). Common summer shelters also include pallets, which are shallow excavations that barely cover the tortoise (Auffenberg 1969 *in* Natureserve 2006). Another common summer behavior for desert tortoises is to rest in depressed or compressed vegetation and soil (Natureserve 2006). Desert tortoises often use more than one burrow; one study showed several burrows being used by one tortoise in a week (Natureserve 2006).

Translocation

Translocation is an important technique in the conservation of the desert tortoise. Translocation can be used to supplement existing populations and to create new populations in the desert tortoise's historic range (Tracy *et al.* 2004). Translocation studies have been conducted in Nevada and Utah, involving tracking more than 300 desert tortoises. These studies showed that translocated individuals have the same survivorship rates and reproductive success as resident individuals. Studies also showed tortoises moving abnormally large distances for the first year after their initial translocation. If tortoises were released in atypical desert tortoise habitat, they generally moved until they reached typical habitat (Field 1999 *in* Tracy *et al.* 2004; Field *et al.* 2003 *in* Tracy *et al.* 2004; Nussear 2004 *in* Tracy *et al.* 2004).

Survey Methods

Berry and Nicholson (1984 *in* Tracy *et al.* 2004) developed a transect survey method using total corrected sign to determine relative tortoise density. Corrected sign is a comparison of sign counts from areas of unknown density to areas with both sign counts and estimated densities (Tracy *et al.* 2004). Transects were conducted using this method in 1998, 1999, and 2000 by the BLM. In February 1995, monitoring protocols were reviewed and possible changes in methodology were discussed, including distance sampling. Line distance sampling, which differed from previous methods, was endorsed in June 1999 as the preferred method for range-wide sampling of the desert tortoise (Tracy *et al.* 2004). Line-distance sampling transects were conducted by the USFWS, from 2001 to 2005, in range-wide sampling of desert tortoises within the 14 DWMAs (Averill-Murray 2006). Density of adult tortoises was relatively stable from 2001 to 2005 in the eastern three recovery units (Northeastern Mohave, Eastern Mohave, and Northern Colorado). Density decreased between 2002 and 2003 in the Northern Colorado and Western Mohave recovery units (Averill-Murray 2006) (Table 1).

Table 1: Density estimates (number/km^2) for the Recovery Units identified in the 1994 Recovery Plan per year (Averill-Murray 2006).

Recovery Unit	2001	2002	2003	2004
Northeast Mohave	2.36±0.923	.94±0.533	3.20±0.489	1.44±0.349
Eastern Mohave	3.11±0.847	4.60±0.791	2.74±0.867	5.67±0.767
Eastern Colorado	11.61±1.824	9.28±1.884	4.00±0.773	5.47±0.700
Northern Colorado	8.23±1.445		6.55±1.120	7.17±1.122
Western Mohave	7.81±0.7444	7.98±0.866	5.59±0.493	5.40±0.679

Threats

The desert tortoise has been extirpated or has severely declined from the western and northern parts of its geographic range in California (Antelope, Indian Wells, and Searles valleys) (Jacobson 1994 *in* USFWS 1994). The desert tortoise is subject to multiple threats simultaneously in many parts of its range, so removing a single threat will not increase the population size if other limiting factors remain (Tracy *et al.* 2004).

The major causes for decline of the desert tortoise are habitat destruction, degradation, and fragmentation from urban and agricultural development; livestock grazing; mining; invasion of nonnative plants; and off-road vehicle (ORV) use (Jacobson 1994 *in* USFWS 1994). Direct mortality or injury of desert tortoises caused by humans and disease are other major threats to the species (Tracy *et al.* 2004).

Agricultural developments cause widespread reduction of the water table, increase raven populations, clear native vegetation, introduce pesticides and fertilizers to habitat, and provide a seed source for nonnative plants (USFWS 1994). For example, Russian thistle seeds have blown from adjacent agricultural fields at Cantil into the Desert Tortoise Natural Area in eastern Kern County, California, where they have become established (BLM and CDFG 1988 *in* USFWS 1994).

Grazing can result in mortality of individual tortoises or eggs, promote soil erosion, damage soil crusts, reduce native vegetation, trample burrows, and increase rate of nonnative species invasion (Jacobson 1994 *in* USFWS 1994). The reduction of native perennial grasses reduces forage availability and protein available to desert tortoises (NatureServe 2006). Livestock grazing has contributed to the reduction of perennial grasses in the genera *Bouteloua, Hilaria, Stipa, Oryzopsis, Poa, Muhlenbergia,* and *Sporobolus,* and perennial shrubs such as *Acamptopappus sphaerocephalus, Lycium andersoni, Grayla spinosa, Ceratoides lanta,* and *Machaeranthera tortifolia* (Bentley 1898 *in* USFWS 1994; Frenkel 1970 *in* USFWS 1994; Humphrey 1958, 1987 *in* USFWS 1994; Humphrey 1987 *in* USFWS1994; Rowlands unpublished BLM 1980 *in* USFWS 1994; USFWS 1994). Livestock grazing has contributed to the spread of nonnative plants, such as *Erodium cicatarium, Schismus barbatus, Schismus arabicus, Bromus species,* and *Salsola iberica* (Kay *et al.* 1988 *in* USFWS).

Fire is a threat to the desert tortoise. Nonnative, ephemeral plants have invaded the Mohave and Colorado deserts. Continuous patches of these plants, such as red brome (*Bromus rubens*)**,** can carry fires over large regions and have caused an increase in high-intensity, large acreage fires. These fires kill fire-intolerant, native annuals and perennials, which are often replaced by fire-tolerant, nonnative species. Fires also fragment desert tortoise habitat and kill individual tortoises (Jacobson 1994 *in* USFWS 1994). Fires are most hazardous to tortoises when they occur during the active season for tortoises. Previously rare, frequency of spring fires is now on the increase due to the encroachment of nonnative species (Brooks 1998 *in* Boarman 2002b). Fire records from 1989-2001 showed that the largest percentage of land was burned in the Northeastern Mojave Recovery Unit (12.6%) and the Upper Virgin Recovery Unit (5.0%) (Brooks 2006).

Freeways, highways, paved roads, dirt roads, and railroads pose a threat to this species (USFWS 1994). Desert tortoise populations are depleted up to a mile or more on either side of roads when average daily traffic is greater than 180 vehicles (Nicholson 1978 *in* USFWS 1994). Dirt roads, which do not get much vehicle use, can cause depression in desert tortoise populations (Berry *et al.* 1986 *in* USFWS 1994). Tortoises can get caught in railroad tracks and overheat or get crushed by a train (U.S. Ecology 1989 *in* USFWS 1994).

Off-highway vehicles (OHV) in the desert tortoise historical range pose a threat to this species (USFWS 1994); OHV activity has increased in recent years in desert habitat. Increased OHV use can have negative impacts, such as tortoises being run over by vehicles, crushing of vegetation, damage to soil crusts, soil erosion, spreading of invasive plants, and increase in fires (USFWS 1994). Recent research in the Mohave Desert has demonstrated that biomass of native vegetation was greater in areas protected from grazing and OHV use than in areas that were unprotected from these activities (Brooks 1995 *in* Tracy *et al.* 2004; Brooks 1999 *in* Tracy *et al.* 2004).

Mining, energy development, utility, and energy facilities in the desert tortoise’s historical range pose a threat to this species (USFWS 1994). These activities cause construction of roads and increased vehicle use, disturbance of soil surface and vegetation, toxic byproducts, refuse of stakes and wire, transfer of title from public lands to private use, fragmentation of habitat, increased habitat for predatory birds, and creation of trenches that tortoises can fall into (USFWS 1994; Olson *et al.* 1992 *in* USFWS 1994; S. Hale, pers. comm. *in* USFWS 1994). Utility and natural gas lines disturb desert tortoise habitat in an area that is 50 to 125 ft wide surrounding the point where the lines are installed (USFWS 1994).

Military activities in the desert tortoise’s historical range pose a threat to this species (USFWS 1994). These activities include construction, operation and maintenance of bases and support facilities (airstrips, roads, etc.); development of support communities; field maneuvers (tank traffic, bombing, testing of explosives, unexploded ordinance littering, shell castings); and chemical distribution.

Diseases, such as Upper Respiratory Track Disease (URTD) and shell disease, are threats to the desert tortoise (Jacobson 1994 *in* USFWS 1994; Berry 1990 *in* USFWS 1990; Avery and Berry *in* USFWS 1990). URTD is caused by the bacterial agent *Mycoplasma agassizii* and is a major cause of tortoise mortality in the Mohave population, particularly in the western Mohave (Berry 1990 *in* USFWS 1994). From 1979 to 1992, total population density in the Desert Tortoise Research Natural Area in the western Mohave decreased by 76%. From 1988 to 1992, this decline was clearly contributable to URTD (Berry 1997). It is probable that pathogenic and nonpathogenic desert tortoise *Mycoplasmas* exist and there is variation among strains of *Mycoplasma agassizii* in their ability to cause URTD (Tracy *et al.* 2004). URTD causes hyperplastic and dysplastic lesions of the upper respiratory tract and clinical signs vary in onset, duration, and severity (Tracy *et al.* 2004). Desert tortoises infected with URTD may show symptoms of clear wet discharges from eyes and nose, loss of weight, and wheezing (Natureserve 2006). Drought and poor nutrition may make tortoises more susceptible to URTD; however, the disease has been documented in healthy tortoises (Jacobson *et al.* 1991 *in* USWFS 1994). URTD may have been introduced to wild populations through illegal releases (Jacobson *in* USFWS 1994). *Mycoplasma* is a horizontally transmissible disease (transmission from one animal to another other rather than from the parent to the offspring) and it may be transmitted by some forms of indirect contact. It is probable that *Mycoplasma* does not persist in burrows of infected tortoises (Tracy *et al.* 2004). A serological test has been developed to confirm the presence of blood antibodies to the URTD pathogen, but no effective cure for the disease is available (Schumacher *et al.* 1993 *in* Natureserve 2006).

Symptoms of the shell disease, cutaneous dyskeratosis (CD), include lesions along scute sutures of the plastron and, to a lesser extent, on the carapace. The disease may be caused by toxins or a nutritional deficiency (Homer *et al.* 1998 *in* Boarman 2002b; Jacobson *et al.* 1994 *in* Boarman 2002b). Herpesvirus was recently identified in desert tortoises and may have population level effects, but very little is known about it (Jacobson *et al.* 1996 *in* Boarman 2002b; Berry 1997 *in* Boarman 2002b).

Direct human mortality, in terms of collecting, shooting, harassing, and killing or injuring with a vehicle, is also a reason for decline of this species (Jacobson *in* USWFS 1994). Individual tortoises have been collected or poached by humans in several radio transmitting studies (Stewart 1991 *in* USFWS 1994; Berry 1990 *in* USFWS 1994). Fifteen percent of 635 dead desert tortoise carcasses from several California studies were wounded by gunshot (Berry 1986 *in* USFWS 1994). Illegal relocations or releasing captive tortoises in the wild poses a threat to native populations from genetic pollution, potential for introducing or spreading disease, and disturbance to the social structure of the host population (USFWS 1994). The outbreak of URTD in the Mohave population appears to be correlated with captive tortoise release sites (Hardenbrook and Tomlinson 1991 *in* USFWS 1994; Jacobson 1993 *in* USFWS 1994; Tomlinson and Hardenbrook 1992 *in* USFWS 1994). Releasing captive tortoises of another species, such as *Gopherus berlandieri,* or another population unit, such as introducing a Sonoran tortoise in the Mohave population, also poses threats to the desert tortoise (USFWS 1994).

Illegal dumping is another threat to the desert tortoise (USFWS 1994). The desert tortoise has been known to eat foreign objects such as rocks, balloons, plastic, and other garbage (John Behler, Chairman of the Freshwater Turtle and Tortoise Group, Species Survival Commission, International Union for the Conservation of Nature and New York Zoological Society, pers. comm., *in* USFWS 1994; Karen Bjorn Dahl pers. comm. *in* USFWS 1994). Objects can be lodged in the gastro-intestinal tract, causing death (USFWS 1994). Balloons that are released in mass are also threats to the desert tortoise (USFWS 1994).

Predators of the desert tortoise include kit foxes, bobcats (*Felis rufus*), coyotes (*Canis latrans*), Gila monsters (*Hypodermal suspected*), golden eagles (*Aquila chrysaetos*), and common ravens (*Corvus corax*) (Turner *et al.* 1987 *in* USFWS 1994; Beck 1990 *in* USFWS 1994; Berry 1985 *in* USFWS 1994; Woodman and Jaurez 1994 *in* USFWS 1994; and Farrell 1989 *in* USFWS 1994). Feral and domestic dogs and cats are also predators of the desert tortoise (Causey and Cude 1978 *in* USFWS 1994; Berry 1979 *in* USFWS 1994). The common raven, whose numbers have increased in the Mohave and Colorado Deserts since 1968, is a major predator of juvenile tortoises (Jacobson 1994 *in* USFWS 1994). Raven populations have increased by nearly 800-1400% in the Mohave and Sonoran deserts over the past 37 years (Boarman and Kristan 2006). Raven population growth rates, dispersal rates, and local abundance continue to be the highest in the west Mohave Desert (Boarman and Kristan 2006). Berry (1990 *in* USWFS 1994) believes that the increase of common raven populations have effected juvenile recruitment. Adults are protected against most predators, except human-caused fatalities (Wilbur and Morin 1988 *in* USFWS 1994; Turner *et al.* 1987 *in* USFWS 1994).

Ectoparasites of the desert tortoise include ticks (*Ornithodoros turicata, Ornithodoros parker*), *Trombicula* mites, and dipteran maggot larvae. Endoparasites and pathogens include intestinal protozoa bacteria, and the oyurate nematode (Morafka *et al.* 1986 *in* Natureserve 2006).

LCR MSCP Conservation Measures

AMM5-Avoid impacts of operation, maintenance, and replacement of hydroelectric generation and transmission facilities on covered species in the LCR MSCP planning area.
To the extent practicable, before implementing activities associated with operation, maintenance, and replacement of hydroelectric generation and transmission facilities, measures will be identified and implemented that are necessary to avoid take of covered species where such activities could otherwise result in take. These measures could include conducting surveys to determine whether covered species are present and, if so, deferring the implementation of activities to avoid disturbance during the breeding season, or redesigning the activities to avoid the need to disturb covered species habitat use areas; staging of equipment outside of covered species habitats; delineating the limits of vegetation control activities to ensure that only the vegetation that needs to be removed to maintain infrastructure is removed; stockpiling and disposing of removed vegetation in

a manner that minimizes the risk of fire; and implementing the BMPs to control erosion when implementing ground disturbing activities.

DETO1-Acquire and protect 230 acres of existing unprotected occupied habitat. Consistent with the mitigation measures identified in the document "Compensation for Desert Tortoise" (Desert Tortoise Conservation Team 1991), the LCR MSCP will acquire and protect 230 acres of unprotected desert tortoise habitat. The acquired habitat will be transferred to an appropriate management agency for permanent protection of species habitat. Although creation of replacement habitat is not considered feasible, protecting existing occupied habitat will ensure that the implementation of covered activities and LCR MSCP conservation measures do not adversely affect the existing distribution, abundance, or population viability of the desert tortoise within the LCR MSCP planning area.

DETO2-Avoid impacts on individuals and their burrows.

1. Before implementing non-flow related covered activities and LCR MSCP conservation measures in desert tortoise habitat, presence or absence surveys will be conducted using approved USFWS survey protocols to locate desert tortoises and their burrows (U.S. Fish and Wildlife Service 1992). The number and location of all tortoises or tortoise sign (e.g., shells, bones, scutes, limbs, scats, burrows, pellets, tracks, egg shell fragments, courtship rings, drinking sites, and mineral licks) that occur within the project area and its zone of influence and whether any tortoises occur outside of the project area whose home range may overlap the project area or its zone of influence should be identified. The project area is defined as any area that will: be cleared or partially cleared; have vehicles on or adjacent to it; be temporarily or permanently used for equipment or materials storage, loading, or unloading; or have its soil or vegetation damaged, fragmented, or disturbed. Desert tortoise presence or absence surveys should be conducted during the typical period of activity for the tortoise (i.e., March 25 to May 31). Surveys should be conducted during the daylight hours. The USFWS considers the results of a presence or absence survey, including the zone of influence, to be valid for no more than 1 year, though the time period may be significantly reduced, depending on project size, location, or proximity to other land disturbance.

2. If desert tortoises are present, the covered activity or LCR MSCP activity will be modified to avoid take of individuals and their burrows. However, if impacts cannot be avoided, clearance surveys will be conducted to locate desert tortoises that will be removed and relocated to other habitat areas. Clearance surveys should be conducted to locate all desert tortoises above and below ground within the project area that would be temporarily relocated or salvaged using the USFWS clearance survey protocol (U.S. Fish and Wildlife Service 1992). Clearance surveys should be conducted immediately prior to surface disturbance at each site within the project area. Surveys should be conducted during daylight hours.

3. If impacts cannot be avoided, desert tortoises should be removed and relocated to other habitat areas, if appropriate. The Desert Tortoise Council guidelines for determining whether tortoises should be moved, mapping tortoise burrows, determining whether burrows should be excavated, finding tortoises in burrows, excavating burrows, constructing artificial burrows, handling tortoise eggs, handling tortoises, processing tortoises, translocating tortoises, and releasing tortoises should be followed (Desert Tortoise Council 1994).

LCR MSCP Research and Monitoring Needs

Conservation measure DETO-1 states that the LCR MSCP will acquire 230 acres of unprotected land. Research needs pertaining to this conservation measure include defining habitat requirements, and threats or impacts to the desert tortoise throughout its historical range. Adequate information is available on these topics to proceed with acquiring land. Conservation measures AMM5 and DETO2 state that impacts to individuals and burrows must be avoided when conducting activities in desert tortoise habitat. The LCR MSCP will follow USFWS protocol for clearance surveys and exclusion fencing, and will follow desert tortoise council protocol for removal and relocation of tortoises, burrows, and eggs (USFWS 1992; Desert Tortoise Council 1994).

Other Potential Research and Monitoring Opportunities

Research needs defined in the Desert Tortoise (Mohave Population) Recovery Plan were as follows: 1) population structure (spatial scale of genetics and demography); 2) long-term analysis of impacts; 3) effectiveness of protective measures; 4) spatial variation in climate and vegetation; 5) nutritional and physiological ecology; 6) reproductive behavior and physiology; 7) restoration, augmentation and translocation; 8) survivorship or recruitment of non-reproductive age classes; and 9) nutritional and physiological ecology of non-reproductive age classes (Tracy *et al.* 2004).

Since the recovery plan was published, there have been numerous papers published on nutritional ecology, reproductive physiology, and the effects of specific impacts on desert tortoise populations (Tracy *et al.* 2004). Few research studies have focused on long-term demography, the effectiveness of recovery actions, translocation, and climatic or vegetative variability. There has been almost no research conducted on epidemiology, long-term impacts on desert tortoise populations, recruitment and survival of non-reproductive age classes, and population and structure of non-reproductive age classes (Tracy *et al.* 2004). Research is needed pertaining to translocation programs; specifically, host-pathogen relations and other community-level interactions (i.e., competition, predator-prey pathogen, pathogen) (Tracy *et al.* 2004). Population/metapopulation dynamics of the desert tortoise needs to be reevaluated (Tracy *et al.* 2005).

Literature Cited

Anderson, M.C., J.M. Watts, J.E. Freilich, S.R. Yool, G.I. Wakefield, J.F. McCauley, and P.B. Fahnestock. 2000. Regression-tree modeling of desert tortoise habitat in the Central Mohave Desert. Ecological Applications 10(3):890-900.

Arizona Game and Fish Department. 2001. Desert Tortoise. Unpublished abstract compiled and edited by the Heritage Data Management System, Arizona Game and Fish Department, Phoenix, AZ. 11 p.

Averill-Murray, R.C., and R.D. Williams. 2006. U.S. Fish and Wildlife Service-Desert Tortoise Recovery Office Update. Abstract at 31st Annual Meeting and Symposium. The Desert Tortoise Council. Tucson, AZ.

Averill-Murray, R.C., M. Brenneman, P. Corn, C. Everly, J.S. Heaton, R. Marlow, P.A. Medica, K.E. Nussear, and R.C. Tracy. 2006. Rangewide Monitoring of the Desert Tortoise: 2001-05 Summary Report: Desert Tortoise Monitoring Committee. Draft Report.

Berry, K.H., 1997. Demographic consequences of disease in two desert tortoise populations in California, USA. Proceedings: Conservation, Restoration and Management of Tortoises and Turtles—An International Conference. New York and Tortoise Society, p. 91-99.

Boarman, W.I. 2002. Desert tortoise (*Gopherus agassizii*). *In:* Boarman, W.I., and K. Beaman, editors. The Sensitive Plant and Animal Species of the Western Mohave Desert. U.S. Geological Survey, Western Ecological Research Center, Sacramento, CA.

Boarman, W.. 2002b. Threats to Desert Tortoise Populations: A Critical Review of the Literature. U.S. Geological Survey Western Ecological Research Center. Sacramento, California.

Boarman, W.I., and Kristan, W.B., 2006. Evaluation of Evidence Supporting the Effectiveness of Desert Tortoise Tecovery Actions: U.S. Geological Survey Scientific Investigations Report 2006-5143, 27 p.

Boarman, W.I., and W.B. Kristan. 2006. Demography of Ravens in the Mohave Desert: Recent Analysis. Abstract at 31st Annual Meeting and Symposium. The Desert Tortoise Council. Tucson, AZ.

Borchard S. 2006. Acquisition of Desert Tortoise Habitat in the California Desert. Abstract at 31st Annual meeting and Symposium. The Desert Tortoise Council. Tucson, AZ.

Brooks, M. 2006. Fire History and Fire Effects in the Mohave Desert. Abstract at 31st Annual meeting and Symposium. The Desert Tortoise Council. Tucson, Arizona.

Bureau of Reclamation. 1996. Description and Assessment of Operations, Maintenance, and Sensitive Species of the Lower Colorado River. Bureau of Reclamation, Lower Colorado Region. Final Biological Assessment Prepared for U.S. Fish and Wildlife Service and Lower Colorado River Multi-Species Conservation Program.

DeFalco, L. 2006. Revegetation of Desert Tortoise Habitat: Creosote Shrublands. Abstract at 31st Annual meeting and Symposium. The Desert Tortoise Council. Tucson, Arizona.

Desert Tortoise Conservation Team. 1991. Compensation for desert tortoise. Unpublished report to the Desert Tortoise Management Oversight Group.

Desert Tortoise Council. 1994. Guidelines for handling desert tortoises during construction projects. Prepared for U.S. Fish and Wildlife Service, Bureau of Land Management, California Department of Fish and Game, Nevada Department of Wildlife, Arizona Game and Fish Department, and Utah Division of Wildlife Resources. San Bernardino, CA.

Dickson S.D., L.S. Hillard and K.A. Naggy. 2006. How well do head-started yearling tortoises survive after releases. Abstract at 31st Annual meeting and Symposium. The Desert Tortoise Council. Tucson, AZ.

Grover, Mark C.; DeFalco, Lesley A. 1995. Desert tortoises (*Gopherus agassizii*): status-of-knowledge outline with references. Gen. Tech. Rep. INT-GTR-316 Ogden, UT: U.S. Department of Agriculture, Intermountain Research Station. 134 p.

Lower Colorado River Multi-Species Conservation Program. 2004. Lower Colorado River Multi-Species Conservation Program, Volume II: Habitat Conservation Plan. Final. December 17. (J&S 00450.00) Sacrament, CA.

NatureServe. 2006. NatureServe Explorer: An online encyclopedia of life [web application]. Version 5.0. NatureServe, Arlington, Virginia. Available http://www.natureserve.org/explorer (Accessed: September 12, 2006).

Tracy, C.R., R. Averill-Murray, W.I. Boarman, D. Delehanty, J. Heaton, E. McCoy, D. Morafka, K. Nussear, B. Hagarty, and P. Medica. 2004. Desert Tortoise Recovery Plan Assessment. USFWS 166 pp plus appendices.

U.S. Fish and Wildlife Service. 1992. Field survey protocol for any Federal action that may occur within the range of the desert tortoise.

U.S. Fish and Wildlife Service. 1994. Desert tortoise (Mohave population) recovery plan. USFWS, Portland, Oregon. 73 pp plus appendices.

BONYTAIL
(*Gila elegans*)

Distribution

Bonytail were historically widespread and common throughout tributaries of the Colorado River and other larger rivers, with historical captures documented from Mexico to Wyoming (Behnke and Benson 1980, Minckley and Deacon 1991, Mueller and Marsh 2002). The first recorded capture of bonytail from the UCRB was by Jordan (1891) with one specimen collected from the Green River. Subsequent historical collections, albeit limited largely to anecdotal and historical fishing creel interviews, in conjunction with limited scientific collection information combine to demonstrate the once-expansive range of bonytail (USFWS 2002). However, during the 1950s bonytail populations began a rather large, yet poorly documented decline in abundance following numerous biotic and abiotic habitat modifications (see below, and as described in the razorback sucker and flannelmouth sucker species profiles). Holden (1991) described the effects of a large-scale rotenone treatment in the upper Green River, while simultaneously providing insight to the rather large population of bonytail present until 1962, at which time a large piscicide treatment occurred in the UCRB. Bonytail numbers were drastically reduced following the closure of Flaming Gorge Dam in 1963, with very few and sporadic captures of bonytail occurring in the UCRB since that time (Vanicek and Kramer 1969, Holden and Stalnaker 1975, Tyus et al. 1982, Valdez 1990).

Bonytail captures in the LCRB follow similar trends. The USFWS (2002) documents an early capture of 16 individuals from the LCRB by R.R. Miller (from the Grand Canyon). Jonez and Sumner (1954) documented a large aggregation of an estimated 500 adults spawning over a gravely shelf in Lake Mohave. During the period between 1976 and 1988, 34 bonytail were captured in Lake Mohave, and some of these fish were incorporated in the establishment of a brood stock, the progeny of which are presently stocked into lakes Mohave and Havasu (Minckley et al. 1989, Minckley et al. 1991, USFWS 2002) and a number of UCRB rivers. Very few wild bonytail captures have been recorded in recent years; therefore, little is known about the specific habitat requirements of this unique species.

Historical Habitat Modifications

Numerous researchers have identified that the major factor contributing to the decline of bonytail and other large-river fishes has been the construction of mainstem dams and the resultant cool tailwaters and reservoir habitats that replaced once-warm, riverine environments (Holden and Stalnaker 1975, Minckley et al. 1991, Mueller and Marsh 2002, USFWS 2002). Competition and predation from nonnative fishes that are successfully established in the Colorado River and its reservoirs have also contributed to their decline (Minckley and Deacon 1991, USFWS 2002). For further detailed information including examples, ramifications, and research needs on the effects of habitat modifications on native Colorado River fishes, please see Tyus (1982), Minckley and Deacon (1991), Mueller and Marsh (2002), and USFWS (2002).

Systematics and Morphomethrics

The following species description is based on information supplied by USFWS (2002):

> Bonytail were first collected from the Zuni River, New Mexico, in 1853, by Baird and Girard during their early expeditions to the Colorado River Basin (Sitgreaves 1853, Girard 1856). *Gila elegans* is commonly known as the bonytail, a name that has been shared by numerous other native chubs of the Colorado River. Bonytail are a streamlined fish, typified by its small head, slender body, and thin, pencil-like caudle peduncle. The head is compressed and the snout overhangs the mouth. Bonytail also have a small, smooth hump (smaller than that of the humpback chub) located directly posterior to the head of adult fish. Bozeck et al. (1984) indicates that bonytail may reach lengths greater than 550 mm, and may weigh over 1100 g. Coloration is typically grey dorsally, fading to white ventrally, with yellowish pigmentation near the base of the pectoral and pelvic fins. Adult spawning fish (males and females) display tuberculation on the head and fins. Dorsal and anal fin rays are typically 10 (Holden 1968, Holden and Stalnaker 1970, Rinne 1976) with caudle peduncle length divided by head length equaling 1.0 (or head length divided by caudle peduncle depth usually being 5.0 or more) (Minckley 1973). Bonytails are mostly scaled throughout the body surface, with 75-88 scales along the lateral line. Scales are not as deeply embedded as those of the humpback chub and the pharyngeal teeth formula is 2,5-4,2. As described by Holden (1968), young bonytail are easily confused with roundtail chubs and humpback chubs, particularly at smaller size classes and in areas of known coexistence. As adults, bonytail are often mistaken due to what appears to be a high level of morphological plasticity among the endemic species of the Colorado River *Gila* complex, and due to understudied levels of introgressive hybridization of the various species of Colorado River *Gila* (Dowling and DeMarais 1993, Douglas et al. 1998). The unique morphology of the bonytail has been hypothesized to be adapted to historical, torrential flows, thought to have been typical of the Colorado River (Miller 1946, Beckman 1963).

Hybridization

As reviewed by USFWS (2002), hybridization between bonytail and other native Colorado River *Gila* species appears to have been common. For example, within the *Gila* complex, inter- and intraspecific morphological variation is apparently extensive where bonytail, roundtail, and humpback chub occur sympatrically. The result of this apparently high degree of hybridization is a relatively high level of phenotypic plasticity, with multiple authors reporting multiple morphologic intergrades present in samples collected throughout the Colorado River (Holden 1968, Holden and Stalnaker 1970, Smith et al. 1979, Douglas et al. 1989, Kaeding et al. 1990, Douglas et al. 1998). Such genetic intermixing was likely common historically and plausibly served to promote phenotypic plasticity and adaptability of the various species to their environment (Dowling and DeMarais 1993). Furthermore, Miller (1946) suggests evidence of species intergrades prior to anthropogenic influences. Recent mitochondrial- and allozyme-based DNA research efforts suggest that bonytail are a uniquely adapted extension of the roundtail chub complex (Dowling and Demarais 1993). The extent of current and ongoing hybridization and its impacts on wild bonytail populations are unknown due to the absence of recent captures, but hybridization and its effects may become important as populations become established through hatchery introductions and overall species recovery, particularly as increasing populations of *Gila* become potentially and increasingly intermixed due to compressed habitat availability (USFWS 2002).

Habitat

Adults

As stated previously, information pertaining to bonytail habitat preferences is very limited due to the extirpation of this species prior to extensive sampling of the Colorado River and its fishery. Limited, early fisheries surveys indicate that the bonytail tended to be found in higher-gradient, gravelly riverine sections, with some degree of habitat use similarities as described for the flannelmouth sucker. For example, bonytail is widely characterized as being adapted to the swifter sections of the Colorado River, with affinity for areas of high flow and rocky habitat. Available information suggests that adult bonytail used fast-water sections, as well as eddies and pool habitats. Vanicek (1967) noted habitat selection of bonytail to coincide with habitats occupied by another native chub, the roundtail chub. Vanicek (1967) found these species in pools and eddies, typically near “fast-flowing” riverine areas but also in slower sections. Holden (1991), citing Flaming Gorge preimpoundment surveys, noted that bonytail were apparently fairly common in the Flaming Gorge area of the upper Green River, a canyon-bound, relatively fast water section of river. Valdez (1990) reported bonytail habitat use as being similar to that of humpback chub, with collections being made in shoreline eddy habitats, boulders, and cobble, and near swift-water sections (in Cataract and Desolation Canyons).

Telemetry studies by Mueller et al. (2003) revealed that adult bonytail prefer interstitial spaces associated with shoreline riprap during daylight hours in Cibola High Levee Pond, whereas open-water areas are more commonly utilized during the nighttime hours. Intensive telemetric surveillance suggests a high degree of site-specific habitat fidelity, with individually marked bonytail consistently returning to the same cavities formed within the riprap type shoreline. These areas may simulate the boulder fields of many of the UCRB canyon areas where bonytail were once common.

Interestingly, a study conducted by Pimentel and Bulkley (1983) suggests that bonytail, when given the opportunity, tend to select water with high levels of total dissolved solids (TDS). Bonytail are able to persist in water with TDS of 4,700 mg/L, the highest tolerance reported for any species of Colorado River *Gila*, suggesting an ability to persist despite anthropogenic water quality and habitat degradation.

Spawning

Bonytail have been documented to spawn over gravel substrates near shore, and were found in water up to 30 feet deep in reservoir situations (Jonez and Sumner 1954). They are hypothesized to use gravel-cobble habitats in lotic environments. Most recently in the LCRB, documentation of successful, natural reproduction in Cibola High Levee Pond suggests that bonytail select shoreline-associated, riprap materials (large-diameter gravel, cobble, and boulder substrates) in water 2-3 m deep for spawning activities (Mueller et al. 2003). Spawning individuals in Lake Mohave display similar diel habitat shifts: adults use in deeper habitats during the day and later form congregations along shoreline habitats (Mueller and Marsh 2002).

Larvae and Juveniles

Relatively little is known about habitat needs for young bonytail. Similar to other native fishes, backwaters and other slackwater habitat types are thought to serve as important nursery areas for young bonytail (USFWS 2002). Larval roundtail and humpback chub tend to use low-velocity backwaters, embayments, and other small, low-velocity habitats along shorelines, moving to water with more current as they become larger (50-75 mm) (Holden 1977, Valdez 1990, Valdez and Ryel 1997). Whether bonytail exhibit the same habitat shift is not known, but it is very likely that the primary reason for the loss of bonytail throughout the basin is related to loss of important nursery habitat. Relatively narrow nursery habitat requirements separate razorback sucker and Colorado pikeminnow from the non-endangered, more common species such as flannelmouth sucker and roundtail chub. Therefore, it seems likely that in a riverine situation bonytail may have a nursery habitat requirement that has not as yet been fully explained.

Young bonytail were most commonly associated with areas of dense overhead cover in depths greater than 1 m. They displayed schooling in warm, shallow areas of Cibola High Levee Pond (Mueller et al. 2003). These findings suggest that refugia-type backwaters designed for bonytail should have similar components in terms of riprapped shoreline materials, one of the few specific habitat preferences that have been documented to date.

Reproduction

Vanicek and Kramer (1969) documented the last substantial spawning of a wild, riverine population of bonytail in Dinosaur National Monument. Ripe fish were collected from mid-June through early July in water temperatures around 18°C. Bonytail estimated as between 5 and 7 years old were found to be ripe (Vanicek 1967), whereas in controlled hatchery environments, Hamman (1985) found bonytail to begin maturing sexually at age 2. Johnston (1999) classified bonytail as being broadcast spawners and suggested that loss of eddy habitat types due to the construction of impoundments may contribute to the apparent reproductive failure of a closely related species, the humpback chub. Marsh (1985) reported bonytail eggs to be adhesive and to apparently remain so throughout the incubational period, which is thought to be an adaptive strategy to swift-moving currents of the mainstem Colorado River.

Ripe bonytail have also been collected from lentic, reservoir situations. As stated previously, Jonez and Sumner (1954) reported active spawning of a large (approximately 500 individuals) aggregate of bonytail in Lake Mohave. Eggs were described as being adhesive, and one individual female contained more than 10,000 eggs, suggesting a high level of fecundity, a trait that appears to be typical for native, Colorado River endemics (see razorback sucker and flannelmouth sucker species profiles). Even higher levels of fecundity were found in hatchery settings, with individual female egg production averaging more than 25,000 eggs per female (Hamman 1982). Spawning bonytail in Cibola High Levee Pond were observed utilizing shoreline riprap materials, typically in mid-April and frequently during nighttime hours, in water temperatures ranging from 20.4 to 21.6°C. They were observed consuming their own gametes, as well as young razorback sucker larvae (Mueller et al. 2003).

Bonytail egg survival appears to be highly influenced by incubation temperature. Hamman (1982) found 90% survival at water temperatures of 20-21°C, 55% survival at 16-17°C, and only 4% survival when temperatures were held between 12 and 13°C. Incubation periods ranged from 99 hours to nearly 500 hours, depending upon water temperatures. Newly hatched fry averaged 6.8 mm (Hamman 1982) in length. This research is corroborated by Marsh (1985), who found bonytail embryos to have the highest survival rates at temperatures near 20°C and indicated that newly hatched larvae averaged 6.0-6.3 mm in size.

Diet

Bonytail diet comprises a wide variety of aquatic and terrestrial insects, worms, algae, plankton, and plant debris (Mueller and Marsh 2002). This is corroborated by McDonald and Dotson (1960) and Vanicek (1967), who found Colorado River chub to feed omnivorously. More quantitative descriptions of bonytail diet preferences are not available, including shifts in diet composition by life stage, with the exception of information from bonytail stocked into Cibola High Levee Pond. This experimental population fed omnivorously, with adult bonytail consuming algae, vegetative material, small fish, and crayfish (*Procambarus* and *Orcopectes* spp.). Young bonytail were documented to feed near the surface of the pond, with gut analysis demonstrating that smaller size classes typically fed on zooplankton and invertebrates (Mueller et al. 2003). More detailed knowledge is unavailable, likely due to the overall rarity of the species.

Age and Growth

Little detailed information exists on naturally recruited bonytail age and growth patterns. According to a USFWS (2002) review, the only substantial findings regarding bonytail age and growth are those reported by Vanicek (1967), who aged 67 bonytail using scales and found the largest to be 7-years-old at a length of 338 mm and weight of 422 g. Ulmer (1983) used otoliths to determine that two Lake Mohave bonytail were 32- and 39-years-old. Rinne (1986) estimated four Lake Mohave fish to be between 34- and 49-years-old. Data suggest that bonytail are typically captured between 338 and 535 mm TL (USFWS 2002). In any case, bonytail are long-lived, a trait that has been speculated by many researchers to be an adaptation to an extremely harsh, unpredictable environment (Mueller and Marsh 2002).

Literature Cited

Beckman, W.C. 1963. Guide to the fishes of Colorado. Colorado State Museum Leaflet 11:1-110.

Behnke, R.J., and D.E. Benson. 1980. Endangered and threatened fishes of the upper Colorado River Basin. Bulletin 503A, Cooperative Extension Service, Colorado State University, Fort Collins, Colorado.

Bozek, M.A., L.J. Paulson, and J.E. Deacon. 1984. Factors affecting reproductive success of bonytail chubs and razorback suckers in Lake Mohave. Bureau of Reclamation, Final Report, Boulder City, Nevada.

Douglas, M.E., W.L. Minckley, and H.M. Tyus. 1989. Quantitative characters, identification of Colorado River chubs (*Cyprinidae*: genus *Gila*) and "the art of seeing well." Copeia 1993:334-343.

Douglas, M.E., R.R. Miller, and W.L. Minckley. 1998. Multivariate discrimination of Colorado Plateau *Gila* spp.: the art of seeing well revisited. Transactions of the American Fisheries Society 127:163-173.

Dowling, T.E., and B.D. DeMarais. 1993. Evolutionary significance of introgressive hybridization in cyprinid fishes. Nature 362:444-446.

Girard, C. 1856. Researches upon the cyprinoid fishes inhabiting the fresh waters of the United States of America, west of the Mississippi Valley, from specimens in the museum of the Smithsonian Institution. Academy of Natural Science of Philadelphia Proceedings 8:165-213.

Hamman, R.L. 1982. Induced spawning and culture of bonytail chub. Progressive Fish Culturist 44:201-203.

Hamman, R.L. 1985. Induced spawning of hatchery-reared bonytail. Progressive Fish Culturist 47:239-241.

Holden, P.B. 1968. Systematic studies of the genus *Gila* (Cyprinidae) of the Colorado River Basin. M.S. Thesis. Utah State University, Logan. 68 p.

Holden, P.B. 1977. A study of the habitat use and movement of the rare fishes in the Green River from Jensen to Green River, Utah, August and September, 1977. Final Report for Western Energy and Land-use Team, Contract 14-16-0009-77-050, BIO-WEST, Logan, Utah.

Holden, P.B. 1991. Ghosts of the Green River: Impacts of Green River poisoning on management of native fishes. Pages 43-54 *In:* W.L. Minckley and J.E. Deacon (editors). Battle against extinction: Native fish management in the American Southwest. University of Arizona Press. Tucson. 517 p.

Holden, P.B., and C.B. Stalnaker. 1970. Systematic studies of the genus *Gila*, in the Upper Colorado River Basin. Copeia 1970(3):409-420.

Holden, P.B., and C.B. Stalnaker. 1975. Distribution of fishes in the Dolores and Yampa River systems of the upper Colorado Basin. Southwestern Naturalist 19:403-412.

Johnston, C.E. 1999. The relationship of spawning mode to conservation of North American minnows (*Cyprinidae*). Environmental Biology of Fishes 55:21-30.

Jonez, A., and R.C. Sumner. 1954. Lakes Mead and Mojave investigations: A comparative study of an established reservoir as related to a newly created impoundment. Federal Aid to Fisheries Restoration Project Completion Report, F-1-R, Nevada Fish and Game Commission, Reno. 186 p.

Jordan, D.S. 1891. Report of explorations in Utah and Colorado during the summer of 1889, with an account of fishes found in each of the river basins examined. Bulletin of the U.S. Fish Commissioner 9:1-40.

Kaeding, L.R., B.D. Burdick, P.A. Schrader, and C.W. McAda. 1990. Temporal and spatial relations between the spawning of humpback chub and roundtail chub in the upper Colorado River. Transactions of the American Fisheries Society 119:135-144.

Marsh, P.C. 1985. Effect of incubation temperature on survival of embryos of native Colorado River fishes. Southwestern Naturalist 30:129-140.

McDonald, D.B., and P.A. Dotson. 1960. Fishery investigations of the Glen Canyon and Flaming Gorge impoundment areas. Utah Department of Fish and Game Information Bulletin 60-3:1-70.

Miller, R.R. 1946. *Gila cypha*, a remarkable new species of cyprinid fish from the Colorado River in Grand Canyon, Arizona. Journal of the Washington Academy of Science. 36(12):409-413

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

Minckley, W.L., D.G. Buth, and R.L. Mayden. 1989. Origin of brood stock and allozyme variation in hatchery-reared bonytail, and endangered North American cyprinid fish. Transactions of American Fisheries Society 118:131-137.

Minckley, W.L., and J.E. Deacon. 1991. Battle against extinction: Native fish management in the American West. University of Arizona Press. Tucson. 517 p.

Minckley, W.L., P.C. Marsh, J.E. Brooks, J.E. Johnson, and B.L. Jensen. 1991. Management toward recovery of razorback sucker. Pages 303-357 *In:* W.L. Minckley and J.E. Deacon (editors). Battle against extinction: Native fish management in the American Southwest. University of Arizona Press. Tucson. 517 p.

Mueller, G.A., and P.C. Marsh. 2002. Lost, a desert and its native fishes: A historical perspective of the lower Colorado River. Information and Technology Report USGS/BRD/ITR—2002—0010. U.S. Government Printing Office, Denver, Colorado, 69 p.

Mueller, G.A., P.C. Marsh, D. Foster, M. Ulibarri, and T. Burke. 2003. Factors influencing poststocking dispersal of razorback sucker. North American Journal of Fisheries Management 23 (1):270-275.

Pimentel, R., and R.V. Bulkley. 1983. Concentrations of total dissolved solids preferred or avoided by endangered Colorado River Fishes. Transactions of the American Fisheries Society 112:595-600.

Rinne, J.N. 1976. Cyprinid fishes of the genus *Gila* from the lower Colorado River basin. Wasmann Journal of Biology. 34(1):65-107.

Rinne, J.N., J.E. Johnson, B.L. Jensen, A.W. Ruger, and R. Sorenson. 1986. The role of hatcheries in the management and recovery of threatened and endangered fishes. Pages 271-285 *In:* R.H. Stroud (ed.). Fish culture in fisheries management. American Fisheries Society, Bethesda, Maryland. Sitgreaves, L. 1853. Report of an expedition down the Zuni and Colorado Rivers. 32nd Congress, 2nd Session, Executive No. 59, Washington, D.C.

Smith, G.R., R.R. Miller, and W.D. Sable. 1979. Species relationships among fishes of the genus *Gila* in the upper Colorado drainage. Pages 613-623 *In:* R.M. Linn (editors). Proceedings of the First Conference on Scientific Research in the National Parks. U.S. Department of the Interior, National Park Service. Transactions and Proceedings Series No. 5.

Tyus, H.M., B.D. Burdick, R.A. Valdez, C.M. Haynes, T.A. Lytle, and C.R. Berry. 1982. Fishes of the upper Colorado River Basin, distribution, abundance, and status. Pages 12-70 *In:* Miller, W.H., H.M. Tyus, and C.A. Carlson (editors). Fishes of the upper Colorado River System: Present and future. American Fisheries Society, Bethesda, Maryland. 131 p.

Ulmer, L. (ed.). 1983. Endangered Colorado River fishes newsletter. California Department of Fish and Game, Blythe.

[USFWS] U.S. Fish and Wildlife Service. 2002. Bonytail (*Gila elegans*) recovery goals: Amendment and supplement to the bonytail chub recovery plan. U.S. Fish and Wildlife Service, Mountain-Prairie Region 6, Denver, Colorado.

Valdez, R.A. 1990. The endangered fish of Cataract Canyon. Final Report of BIO-WEST, Inc., Logan, Utah, to Bureau of Reclamation, Salt Lake City, Utah.

Valdez, R.A., and R.J. Ryel. 1997. Life history and ecology of the humpback chub in the Colorado River in Grand Canyon, Arizona. Pages 3-31 *In:* C. van Riper, III, and E.T. Deshler (editors). Proceedings of the Third Biennial Conference of Research on the Colorado Plateau. National Park Service Transactions and Proceedings Series 97/12.

Vanicek, C.D. 1967. Ecological studies of native Green River fishes below Flaming Gorge Dam, 1964-1966. Ph.D. Thesis, Utah State University, Logan. 125 p.

Vanicek, C.D., and R. Kramer. 1969. Life history of the Colorado squawfish, *Ptychocheilus lucius*, and the Colorado chub, *Gila robusta*, in the Green River in Dinosaur National Monument, 1964-1966. Transactions of the American Fisheries Society 98:193-208.

RAZORBACK SUCKER
(*Xyrauchen texanus*)

Distribution

Razorback sucker were historically widespread and common throughout the larger rivers of the Colorado River Basin, from Sonora and Baja, California, into Arizona, Colorado, Nevada, New Mexico, and Wyoming (Minckley et al. 1991, Marsh 1996). Gilbert and Scofield (1898) noted particularly high razorback sucker abundance in the Lower Colorado River Basin (LCRB) near Yuma, Arizona; however, Bestgen (1990) indicates that razorback sucker may have historically been uncommon in the turbulent canyon reaches of the LCRB, citing research by Tyus (1987) and Lanigan and Tyus (1989) that suggests that razorback sucker in the Green River (the largest known riverine population) were typically found in calm, flatwater river reaches, not turbulent, fast-water canyon reaches. This trend is evident even within basins, as razorback sucker are typically collected in sand-bottomed, low-gradient, flatwater reaches outside of the spawning period. Razorback sucker have persisted in several of the reservoirs that were constructed in the LCRB; however, these populations were composed primarily of adult fish that apparently recruited during the first few years of reservoir formation (Bestgen 1990). Residual lacustrine populations of long-lived adults then disappeared 40 to 50 years following reservoir creation and the initial recruitment period following reservoir creation (Minckley 1983, McCarthy and Minckley 1987). The largest reservoir population, estimated at 75,000 in the 1980s, occurred in Lake Mohave in Arizona and Nevada, but it had declined to less than 3,000 by 2001 (Marsh et al. 2003). Today, the Lake Mohave population is largely supported by stocking captive-reared fish (Marsh et al. 2003, Marsh et al. 2005). Catches of razorback sucker were reported often from the early 1940s through early 1980s in the LCRB (Minckley 1983, Marsh and Minckley 1989). More recently, over 12 million razorback sucker have been stocked into the LCRB with limited success in retention and survival (Mueller et al. 2003).

To date, the only substantial natural razorback sucker recruitment (low, yet steady numbers) and documentation of razorback sucker progression through all life stages in the LCRB occurs in Lake Mead, with limited and sporadic captures of naturally occurring fish throughout the remainder of the LCRB (Marsh and Minckley 1989; Holden et al. 1997, 1999, 2000a, 2000b, 2001; Abate et al. 2002; Welker and Holden 2003; Welker and Holden 2004).

Historical Habitat Modifications

Numerous researchers have identified that the major factor contributing to the decline of razorback sucker and other large-river fishes has been the construction of mainstem dams and the resultant cool tailwaters and reservoir habitats that replaced a once-warm, dynamic, riverine environment (Holden and Stalnaker 1975, Joseph et al. 1977, Wick et al. 1982, Minckley et al. 1991). This change in the physical environment presumably allowed for an increase in competition and predation from nonnative fishes, which are successfully established in the Colorado River and its reservoirs and have also contributed to native fish population declines (Minckley et al. 1991). For further detailed information including examples, ramifications, and research needs pertaining to the effects of habitat modifications on native Colorado River fishes, see U.S. Fish and Wildlife Service (USFWS 1998, 2002), Minckley et al. (1991), Tyus (1990), and Tyus and Karp (1990).

Systematics and Morphomethrics

Xyrauchen is one of three monotypic genera of the family Catostomidae. According to Bestgen (1990) and the USFWS (1998), Abbott (1861) originally described the razorback sucker as *Catostomus texanus*. Subsequent classifications were made by Kirsch (1889), Jordan (1891), Hubbs and Miller (1953), LaRivers (1962), and Minckley (1973). Meristic and morphological descriptions given by Abbot (1861), Ellis (1914), Hubbs and Miller (1953), Minckley (1973, 1983), Moyle (1976), Snyder and Muth (1990), and McAda and Wydoski (1980), as cited in Bestgen (1990) follow below:

> The razorback sucker is distinguishable from all other catostomids by its unique, abruptly rising, bony, dorsal keel rising posterior from the head. Body shape is elongate, robust, and somewhat laterally compressed. The caudle peduncle tends to be short and deep. An enlargement of the interneural bones forms the distinctive razor-like keel, providing basis for the common name, razorback sucker. The moderate sized mouth has a clefted lower lip, and lateral margins of the lips are continuous and rounded. Razorback sucker have elongated heads with a flattened dorsal surface and well developed fontanelle. Primary dorsal fin rays are usually 1415, primary anal fin rays 7, vertebrae 45-47, scales in the lateral series range from 68-87, with gill rakers containing 44-50 on the first arch. Body coloration is dark brown to olivaceous on the upper dorso-lateral surfaces and ranges from yellow to white on the lower ventro-lateral surfaces. Adults can reach up to 1,000 millimeters (mm) total length (TL) and weigh 5-6 kg, but are more typically found within the 400-700 mm TL range, weighing less than 3 kg. During spawning, razorback suckers are sexually dimorphic, with breeding males showing bright yellow and orange laterally and ventrally, dark dorsal surfaces, and tuberculation present, especially on the anal and caudle fins.

Furthermore, Eastman (1980) described razorback morphology, based on skeletal measurements, as being heavily ossified, thickened, and likely adapted to the strong river currents historically occupied by this species. Larval stages are best described in Snyder et al. (2004).

Hybridization

As reviewed by Bestgen (1990), hybridization between razorback sucker and other native Colorado River catostomid species has historically been documented to occur. Most often, razorback sucker have been shown to hybridize with flannelmouth sucker, but they may also hybridize with Sonora suckers (*Catostomus insignis*), and other native catostomids (Hubbs et al. 1943, Hubbs and Miller 1953, Holden 1973, Holden and Stalnaker 1975, McAda and Wydoski 1980, Minckley 1983, Bozek et al. 1984, Tyus and Karp 1990, Douglas and Marsh 1998). Buth et al. (1987) used allozymic data to directly quantify presumed introgression in the range of 0-5% toward flannelmouth sucker and 0-3% toward razorback sucker. Furthermore, in a natural river setting, Ryden (2000) noted adult flannelmouth sucker were captured consistently over the same cobble-bottomed riffles as mature, adult razorback sucker, suggesting concern for possible hybridization in San Juan River populations due to an overlap in physical habitat usage of the adult life stage of both species.

Habitat

Adults

Historically, razorback sucker inhabited virtually all components of riverine habitat; in particular, low-velocity habitats such as backwaters, sloughs, oxbow lakes, and other slackwater habitats within the main channel were important for razorback sucker (Holden 1973, Holden and Stalnaker 1975, Behnke and Benson 1980, Minckley 1983). Seasonally submerged off-river habitats, including bottomlands and other marsh-like, lowland habitats, may have also been important habitat for razorback sucker prior to the construction of mainstream dams and the resultant changes in flow regimes, especially during spring-runoff periods (Tyus and Karp 1989, Bestgen 1990, Osmundson 2001).

More recent authors have documented that habitat selection by adult razorback sucker changes seasonally. Tyus and Karp (1990) document habitat use by adult razorback sucker to consist of flooded areas during spring months. Radiotelemetry efforts by Tyus (1987) identified adult fish utilizing near-shore runs during the spring, but they subsequently shifted habitat use during the summer to shallow waters associated with submerged mid-channel sandbars, with little use of backwaters. This suggests that the use of backwaters by razorback sucker may be overstated and an artifact of relatively easy capture with electrofishing rather than actual habitat use and preference. Osmundson and Kaeding (1989) reported adult razorback using pools and slow eddies from November through April, shifting to runs and pools from July through October. They also note increased backwater habitat use by adult fish during the months of May and June, the typical UCRB spawning period.

More detailed information of razorback sucker habitat use, needs, and selection is provided by Ryden (2000), based on radio-telemetered razorback sucker occupying the dynamic and relatively natural (by today’s standards) San Juan River of the UCRB. During pre-runoff periods (March and April) tagged fish were found to use a variety of low-velocity habitats. Habitat usage included pools, eddies, shoals, and backwaters, with evident seasonal use of fast-water habitat types. Ryden (2000) indicates that the majority of these habitats were located along the inner edge of large bends in the main river channel. Specific habitats selected for during the month of March were primarily considered to be slow or slackwater habitat types, with the most highly selected habitat type being pools. In March the mean water depth at fish contact locations was 2.7 feet (ft), with warmer temperatures at razorback sucker locations than in adjacent main-channel habitats (mean = 10.9°C, main channel = 9.8°C). Mean bottom velocity in March was 1.5 feet per second (ft/s), while mean water column velocities averaged 1.7 ft/s. During April, razorback sucker primarily selected low-velocity, sand-shoal habitats, as well as other backwater and pool areas. April was reported to be the only month of the year in which sand-shoal or backwaters were the most commonly selected habitat types. Furthermore, in April, mean water depth at fish location was 2.3 ft, with razorback sucker seeking warmer temperatures (13.0°C) than the main channel (12°C). Mean bottom velocity was found to be 0.6 ft/s, and average column velocity was 1.0 ft/s. During May, habitat selection demonstrated that razorback sucker showed a strong preference for eddy habitats located along the inside of large river bends. Also during May, razorback sucker displayed a strong affinity for mid-channel cobble riffles and run-riffles, as well as shoreline cobble-shoal, run-type habitats. Fish collected in these areas appeared to be exhibiting spawning behavior coinciding with the ascending limb of the hydrograph (see spawning ecology section below). Mean water depth usage in May was 3.3 ft, and temperatures in habitats utilized by razorback sucker were the same as those recorded for the main channel (14.8°C), with bottom velocities averaging 0.8 ft/s and water column velocities averaging 1.4 ft/s.

During runoff, or the descending limb of the hydrograph, and post-runoff months (June and July), razorback sucker habitat selection in the San Juan River was dominated by use of inundated vegetation. During high-flow periods radio-telemetered razorback sucker were found utilizing the river's margins and other low-velocity areas. Ryden (2000) suggests that habitat selection in June was likely the result of fish avoiding high, turbulent flows, as well as foraging forays. Water depths utilized in June averaged 3.9 ft, and June was the last of three consecutive months where water temperatures at fish locations were warmer than adjacent main-channel areas (15.0°C versus 14.8°C). Mean bottom velocity at the June contact locations was 1.7 ft/s, while the water column velocities were 2.0 ft/s. Habitat use during July, as flows began to recede, was reported to be very similar to the habitat use described during May, with eddies being the dominant habitat type utilized. Ryden (2000) reports the mean bottom velocity during July to be 0.7 ft/s, mean column velocity to be 1.6 ft/s, and the average temperature occupied by razorback sucker to be 21.1°C.

During the post-runoff summer and fall months (August through October) Ryden (2000) found razorback sucker displaying unique habitat selection, compared with the periods mentioned earlier. For example, a strong shift of habitat types, from slow-water to main-channel and fast-water habitats, was noted. No low-velocity habitat types were selected for during the summer-fall base-flow period. For example, during August, razorback sucker were typically found utilizing main-channel runs and shoal-runs. Likewise, depths utilized by razorback sucker tended to increase, with the mean depth of locations being 6.2 ft. During September similar habitat use was observed, and in October tagged fish were only observed using main-channel runs with mean water depths of 4.0 ft. These spring-to-summer habitat shifts in the San Juan River are similar to the Green River razorback sucker habitat shifts seen by Tyus and Karp (1989).

Habitat selection during the fall-winter transitional period (November) resulted in fish being located only in mid- and main-channel run habitats (Ryden 2000). Mean water depth at fish location was 3.8 ft, and mean temperature at fish location was reported as 5.3°C. Mean bottom velocity at fish locations in November was 1.2 ft/s, while the mean column velocity was 1.7 ft/s.

During winter base-flow periods (December through February) only two habitat types were selected. Main-channel runs and edge pools were selected for during early December, when daytime water temperatures surpassed 3.0°C. However, later in December, as temperatures began to decline, radio-tagged razorback sucker were observed utilizing edge pools only, and fish became notably more sedentary. Mean temperatures throughout the river were 3.0°C, and velocities averaged 1.3 ft/s on the bottom of the river and 1.5 ft/s higher in the water column. In January razorback sucker were only found utilizing edge pools. They only ventured from these pools when water temperatures rose above 3.0°C and then only for very short time periods. Mean temperatures throughout the river were 1.3°C, with mean bottom velocities of 0.5 ft/s and mean column velocities of 0.6 ft/s (Ryden 2000).

During February tagged razorback sucker once again became fairly active and selected edge pools, main-channel runs, eddies, and shore runs. Water depth at fish locations averaged 3.7 ft. Mean velocity at point of contact was 1.0 ft/s on the bottom, as well as at mid-column. Ryden (2000) reports the mean temperature at point of razorback sucker contact to be identical to that of adjacent main-channel habitats, 4.3°C (Ryden 2000).

Comparing the specific findings of Ryden (2000) with findings of researchers in other UCRB locations, similar trends of razorback sucker habitat use are evident. For example, water velocity selection by adult razorback sucker is also typified by seasonal shifts in preferences. Tyus (1987) noted that during the summer, razorback sucker typically were found utilizing velocities averaging 0.5 m/s, while in the winter months adult fish were typically found in currents moving 0.03-0.33 m/s. These findings corroborate hypotheses and findings of Lanigan and Tyus (1989) and Minckley et al. (1991) that few adult razorback sucker utilize swift, whitewater habitats (e.g., Marble and Grand canyons of the LCRB), although radio-telemetry investigations have documented movement of fish through these locations (Tyus and Karp 1990). Furthermore, it becomes apparent that razorback sucker in a natural river setting do not appear to utilize solely backwater habitat types, although it appears that these habitats are important during specific times of the year. Lastly, adult razorback sucker have been reported to select shallower depths during the summer months (0.9-1.65 m) while typically utilizing deeper depths during the winter months (1.65-2.16 m) (Osmundson and Kaeding 1989).

In contrast, hatchery-raised, sonic-tagged razorback sucker in the LCRB were found to use backwater habitat types more frequently, in relationship to their availability, compared to other, main-channel habitat types in the LCRB throughout every season of the year (Bradford et al. 1998, Bradford and Gurtin 2000). However, in contrast, Lee (2005) found that further telemetric investigations in the LCRB show that adult fish prefer main-channel habitats, as virtually all radio-telemetry contacts made with fish were in the main-channel areas typically associated with eddies and other slow-moving, near-shore, sand depositional habitats, not backwaters. This is very recent information, and the reasons for the difference in habitat usage are still being studied. More specific to findings presented in Bradford et al. (1998) and Bradford and Gurtin (2000), Slaughter et al. (2002) reports that adult razorback sucker prefer large, irregularly shaped backwaters with a mean depth greater than 1.5 meters (m). Backwater size and depth were found to be more important in determining LCRB razorback sucker habitat usage than were water quality factors such as turbidity, pH, or temperature (Slaughter et al. 2002). Mueller (1989) observed spawning razorback sucker in LCRB riverine habitat. This habitat was a main-channel, backwater interface at the mouth of a dry wash, and substrates consisted of scoured sands and gravels. Habitat depths were between 3.9 and 6.6 ft, and water velocities were reported between 0.0 and 1.2 ft/s.

Discrepancies in annual habitat-use findings within and between the UCRB and the LCRB have been attributed to a general lack of contacts with fish, particularly in the LCRB, but more likely are thought to reflect dramatic differences in habitat availability between the UCRB and the LCRB. For example, as previously stated, UCRB reaches tend to consist of higher-gradient, erosional, dynamic sections compared with the more depositional, channelized, homogenous habitats types that are occasionally interspersed with highly vegetated, perennial, and permanently connected off-channel backwater impoundment structures typical of the LCRB (Bradford and Gurtin 2000). Lastly, based on observed habitat use in the UCRB (a more natural riverine environment), it can be speculated that the habitat preferences reported for razorback sucker in the LCRB (i.e., mainly backwater habitat use) may simply be a reflection of habitat availability in this highly altered system. It appears as though razorback sucker, although displaying extensive use of backwaters in the LCRB, may be actually (or simply) using the best available habitat, not by preference, but potentially by necessity.

One of the current habitat types presently occupied by populations of razorback sucker are lentic areas imposed by various impoundments in the LCRB. In these lentic areas, adult razorback suckers have also been documented to display interesting and rather extensive habitat use. The majority of such information suggests that lentic-dwelling razorback sucker use a wide variety of habitats, including vegetated areas, littoral shoreline habitats, and substrates ranging from silt and sand to gravel and cobble. Adult razorback sucker have been documented via sonic surveillance to typically occupy depths less than 30 m (averaging between 3.1 and 16.8 m) and are generally located within 50 m from the shore during winter months (less than 30 m from shore during peak spawning activity). However, during summer months, adults were located at deeper depths, often surpassing 30 m, in an effort to hold body temperatures between 18 and 22°C, a behavior thought to maximize bioenergetics (Marsh and Minckley 1989, Holden et al. 1997, 1999, 2000a, 2000b, 2001, Mueller et al. 2000, Abate et al. 2002, Welker et al. 2003, Welker and Holden 2004). Thermal preferendum for adult razorback sucker was estimated to lie within the range of 22 to 25°C based on laboratory observations (Bulkley and Pimentel

1983). Information on the pH preferences specific to razorback sucker were not found, but warm-water fish species, in general, survive well within a pH range of 6.5 to 9.0 (Boyd 1979, Piper et al. 1982). Furthermore, the majority of backwaters investigated by Slaughter et al. (2002) ranged between a pH of 8 and 9. Boyd (1979) and Piper et al. (1982) also suggest that fish growth may become hindered when dissolved oxygen concentrations drop below 6.0 milligrams per liter (mg/L). However, this may not be the case for razorback sucker, as early life stages have been reported utilizing backwater habitats with dissolved oxygen levels approaching 2.0 mg/L at times, with critical dissolved oxygen levels dependant upon water temperatures (Modde 1996, Modde et al. 2001).

Spawning

The spawning season for razorback sucker has been reported to begin as early as November in some LCRB reservoirs and to continue through June in some populations of the UCRB. In upper basin riverine habitats, ripe razorback sucker have been collected from mid-April to mid-June, typically over a very limited time frame (4-5 weeks) (Tyus 1987, Osmundson and Kaeding 1989, Tyus and Karp 1989, 1990, Bestgen 1990). However, in lentic lower basin habitats the majority of spawning generally is carried out between January and April, months when water temperatures are typically within the range of 10-15°C (Bestgen 1990). Male razorback sucker remain ripe for a period of 2-28 days, while females apparently are ripe for less time (2-15 days) in the Green River (Tyus and Karp 1990) but appear to have extended periods of sexual activity in lower basin reservoirs (Holden et al. 2001). Although spawning razorback sucker have been collected over a variety of substrates, the majority of spawning individuals tend to be captured over clean gravel- and cobble-sized substrates (Douglas 1952, Tyus 1987, Bozek et al. 1990, Tyus and Karp 1990, Minckley et al. 1991). In UCRB rivers spawning occurs during the ascending limb of the hydrograph (et al. 2005), which apparently is an important adaptive feature for larvae as discussed below.

In the Green River, when spring flows have elevated to allow access to bottomland and backwater habitats, adult razorback sucker have been documented moving into these slightly warmer than main-channel environments (typically 2-4°C warmer). This behavior has been termed “staging” because it occurs just before and during spawning, and presumably allows for additional heat units to be obtained, a strategy that is thought to stimulate gamete production and minimize the costly act of spawning bioenergetically (Tyus and Karp 1990, USFWS 1998, Holden 1999, Ryden 2000). Razorback sucker have also been documented to use warmer backwater habitat types post-spawn, apparently to recover and feed (Modde and Irving 1998).

Reservoir-spawning razorback sucker have been documented to successfully spawn in various LCRB impoundments. Spawning populations have been located in Lake Mead (Jonez and Sumner 1954, Holden et al. 1997, 1999, 2000a, 2000b, 2001, Abate et al. 2002, Welker et al. 2003, Welker and Holden 2004), Lake Mohave (Bozek et al. 1984, Marsh and Langhorst 1988, Mueller 1989, Bozek et al. 1990), Lake Havasu (Douglas 1952, Minckley 1983), Senator Wash Reservoir (Medel-Ulmer 1980), and likely other locations. Spawning activities are most frequently associated with relatively shallow, flat to gently sloping shoreline areas over relatively clean gravel and cobble (Bestgen 1990). Spawning activity has been documented in depths up to 20 meters in Lake Mead, but typically occurs in less than 2 meters of water (Minckley et al. 1991, Holden et al. 1997, 1999).

Spawning fish have been documented to congregate near river inflow areas that tend to be somewhat more turbid than the majority of the available spawning areas (Jonez and Sumner 1954, Holden et al. 1997, 1999). Most of this spawning in the LCRB results in larvae but little or no recruitment, apparently due to the lack of nursery habitat for young that allows them to escape predation. Recent studies in Lake Mead have shown that spawning of reservoir-recruited fish presently occurs only in a few sites with abundant nearby vegetation and turbidity that serves as cover for the larvae. Apparently, increased turbidity and vegetation serve as cover to promote razorback sucker survival during the highly vulnerable early life stages (Holden et al. 1997, 1999; Johnson and Hines 1999; Holden et al. 2000a, 2000b, 2001; Mueller et al. 2000; Abate et al. 2002; Welker et al. 2003; Welker and Holden 2004). This suggests that although razorback sucker will spawn in a variety of areas, only areas that promote recruitment will result in long-term population survival.

The majority of information on reproduction in lotic systems comes from UCRB research. McAda and Wydoski (1980) collected razorback sucker in spawning condition from gravel bars in water typically 1 m deep. They report substrate used by spawning razorback sucker to consist largely of cobble located in water velocities of approximately 1 m/s. Researchers in the Green and Yampa rivers of the UCRB have depicted that spawning occurs on main-channel gravel and cobble bars (McAda 1977, McAda and Wydoski 1980, Tyus 1987, Tyus and Karp 1990, Modde and Irving 1998). Bliesner and Lamarra (2005) measured substrate size and depth to embeddedness at a suspected razorback sucker spawning site on the San Juan River and compared the information with another nearby riffle. They found that the suspected spawning site had smaller substrate (average of 3.5 cm), deeper depth to embeddedness, and fewer fine materials than the control riffle. This suggests that razorback sucker may have narrower preferences for spawning habitat when given the opportunity to select a site than has been generally known.

Modde and Irving (1998) used radiotelemetry data to document the spawning activity of individually tagged fish at different spawning locations during their 1993-1995 study, suggesting that razorback sucker in the Green River represent a single reproductive population. Tyus and Karp (1990) used radiotelemetry to document the importance of flooded lowlands and other slackwater habitats as resting-feeding areas for razorback suckers during the breeding season. Spawning in riverine sections is associated with increasing spring flows and associated increases in turbidity (Tyus 1987, Tyus and Karp 1990, Modde et al. 2005). Razorback sucker also display an apparently strong spawning site fidelity both in lentic and lotic habitats (Mueller 1989; Tyus and Karp 1990; Holden et al. 2000a, 2000b, 2001; Abate et al. 2002; Welker et al. 2003; Welker and Holden 2004; Modde et al. 2005).

In the LCRB Mueller (1989) provides insight to riverine razorback sucker spawning below Hoover Dam and in the lower Colorado River below Davis Dam. He indicates similar observations as those described above, with razorback sucker typically spawning in water depths between 1.2 and 2.0 m and velocities ranging from 0.00 to 0.37 m/s. Most recently in the LCRB, spawning activities of 126 razorback sucker were visually observed upstream from Needles Bridge, in water approximately 1 m deep over large cobble substrates (Wydoski 2005). This is particularly interesting, as Ryden (2000) and other researchers from the UCRB (e.g., Modde et al. 2005) have associated spawning with large riffle habitats comprising relatively clean cobble substrates, and the area described by Wydoski (2005) is likely one of the few such habitat types in the LCRB. This suggests that razorback sucker populations (and likely other native fish populations) are selecting the best of the limited habitat available for use during the various times of the year and life history stages.

If the multitude of UCRB habitats described by Ryden (2000) and others were available in the LCRB, would we see different habitat use by riverine populations of razorback sucker? In addition, is it plausible that the spawning razorback population described by Mueller (1989) is actually utilizing the only/best in-channel riffle habitat currently available? Are there options for increasing the complexity of the lower river to promote the entire suite of razorback sucker and other native fish life-history stages through various in-channel and off-channel habitat manipulations? It seems plausible at least that populations of native fishes in the LCRB would benefit from research and manipulative efforts directed toward these ends, with the overall goal of restoring habitat that was historically selected for, rather than habitat that is for one reason or another “better” than other habitat types currently available.

Larvae

In lentic (reservoir) settings razorback sucker larvae have been collected over a variety of habitat types, but they typically are collected over or near areas frequented by adult spawning aggregates. As a result, the majority of larval fish are captured over gravel and cobble, at near-shore locations, typically at depths of 0.0-4.9 m (Sigler and Miller 1963; Minckley 1983; Bozek et al. 1984; Marsh and Langhorst 1988; Holden et al. 1997, 1999, 2000a, 2000b, 2001; Abate et al. 2002; Welker et al. 2003; Welker and Holden 2004).

In the Green River larval razorback sucker apparently have an affinity for backwater and flooded bottomland habitats (Tyus 1987, Muth et al. 1998). Historically, high spring flows flooded low-lying areas along the river and redistributed recently emerged and drifting larval razorback sucker into these food-rich backwaters and other seasonally flooded bottomlands, providing unique nursery habitats for razorback sucker (Tyus and Karp 1989, 1990; Modde 1996; Modde et al. 1996; Modde et al. 2005). Laboratory experimentation has documented the importance of backwater habitats for larval razorback sucker by evaluating nocturnal drift tendencies of young razorback sucker exposed to various degrees of flow. Drift tended to increase with an increase in flows, a scenario that would lead to downstream transport, eventually resulting in larval fish being deposited into relatively calm, low-flow environments, conditions present in backwater habitats (Tyus et al. 2000). However, construction of mainstem dams has reduced spring flows and eliminated important nursery areas. Nursery habitats have been either cut-off or do not refill due to insufficient flow in the river. Recently, a “reset” hypothesis has been suggested, which requires the flooded bottomlands to be dried every year or so and then re-flooded, thereby reducing numbers of potential predators in areas that maintain water between years (Modde 2005). Predation in nursery habitats appears to be the major limiting factor for razorback sucker in both the UCRB and LCRB today (Tyus and Karp 1989, Osmundson and Kaeding 1990, Minckley et al. 1991, Mueller 1995, Tyus and Saunders 1996, Modde et al. 2005).

Wild-spawned razorback sucker larvae have been collected in the San Juan River of New Mexico and Utah annually since 1998 (Brandenburg et al. 2005). The larvae are collected with seines in small backwaters, embayments, and other low-velocity habitats along shorelines. The San Juan River floodplain does not have large, flooded bottomlands like the Green River system, suggesting that razorback sucker larvae can survive in the face of nonnative predators without large nursery habitats. As noted below, some larvae in the San Juan River are escaping predation and have been found well into the juvenile stage. These findings are unique in that no other riverine or reservoir system with a host of predators has shown the ability to recruit razorback sucker except for Lake Mead. It also suggests that larval habitat may not be as specific (i.e., flooded bottomlands) as is being studied in the Green River system.

Juveniles

Habitat important to the juvenile life stages of razorback sucker remains relatively understudied, as catches of juveniles remains minimal, presumably due to the predatory and competitory impacts of nonnative species (Tyus 1987, Bestgen 1990, USFWS 1998). The majority of juvenile, riverine catches come from the UCRB (Taba et al. 1965, Gutermuth et al. 1994, Modde 1996), with almost non-existent data on juvenile habitat use from the LCRB. Brandenburg et al. (2005) recently captured wild-spawned juvenile razorback sucker in the San Juan River. They captured 125 juveniles from 30 to 125 mm TL in 2002, and 10 juveniles in 2003. Golden and Holden (2005) captured six wild juveniles in the San Juan River in 2004 ranging in size from 54 to 94 mm standard length (SL). All of the juveniles in the San Juan River were found using seines in shoreline habitats including backwaters, embayments, and other lower-velocity habitats. In addition, Jackson (2005) collected six other wild-spawned juveniles from 120 to 280 mm TL using electrofishing in the lower San Juan River in 2003 and 2004. Habitats for these fish were not recorded, but they were likely also collected from shoreline habitats.

Mueller and Marsh (1998) tracked movements of 55 hatchery-reared subadult (juvenile) razorback sucker that were released into Lake Mohave and Lake Powell. Their telemetry data demonstrated that juvenile razorback sucker utilized backwaters, vegetated areas, and rocky cavities (thought to provide important cover and food resources). This description of habitat use is corroborated by the recapture of two experimentally stocked juvenile razorback sucker in the San Juan River (Holden 1999). These fish were found occupying slackwater and backwater pools 1 to 3 ft deep and 1 to 3°C warmer than adjacent main-channel habitats. Razorback sucker studies in the LCRB on Lake Mead (Holden et al. 1997, 1999, 2000a, 2000b, 2001; Abate et al. 2002; Welker et al. 2003;

Welker and Holden 2004) have documented capture of wild, sexually immature, juvenile razorback suckers. This is one of the only known locations documented to produce this rather obscure life stage with periodic consistency. Juvenile fish were mainly collected near spawning areas with adult fish, although no juveniles showed signs of sexual maturity.

Studies by Modde (1996) and Modde et al. (2001) in the Green River demonstrate that juveniles and subadult razorback sucker may favor floodplain depressions when available (depth of 1-2 m, dissolved oxygen remaining greater than 2.0 mg/L, usually above 5.0 mg/L, and maximum surface temperatures 26.6°C) over main-channel habitats based on habitat variables such as zooplankton density, water temperature, depth, and vegetation abundance. These researchers also indicate that growth and survival in floodplain depressions are more likely than in main-channel habitats despite heavy impacts of nonnative fish predation and competition typically associated with backwater habitats. Furthermore, they suggest that draining wetlands before spring may be important for nonnative fish control, while still allowing razorback sucker growth and development to occur during the remainder of the year. Additionally, Modde (2005) outlines strategies and indicates that a combination of correct flow regimes (to allow for larval razorback sucker deposition into floodplains), coupled with annual reset draining of backwaters (to remove residual nonnative fishes), increases young razorback sucker growth and survival throughout the first year of life by allowing razorback sucker to exist at sizes similar to their nonnative competitors and predators. Modde (1997) documented similar growth and survival rates for young-of-year razorback sucker and carp (*Cyprinus carpio*) in a managed wetland. Mueller et al. (2003) demonstrates that flow acclimation of stocked razorback sucker may be another important way to bolster year-class strength of natural populations, and Marsh et al. (2005) indicate that a size increase of repatriated razorback sucker to lengths greater than 350 mm TL doubles post-stocking survival.

Reproduction

Fecundity for razorback sucker expressed in terms of number of ova per unit standard length was derived by Minckley (1983). Estimates by Minckley (1983) ranged from 1,600 ova/cm SL to 2,000 ova/cm SL. These results were based on 15 fish estimated at having anywhere from 27,614 to 144,000 total number of ova, accounting for 9.2-11.5% of an individual female razorback sucker's body weight. Male gonadal information was not supplied. Bozek et al. (1984) indicated that during the spawning season in Lake Mohave, male:female ratios of razorback suckers ranged from 1.2 to 3.6:1. They also report that approximately 80% of male and less than 65% of female razorback suckers were ripe during peak spawning activity. Current recapture data compiled by Albrecht and Holden (2005) from nine consecutive years of accumulated data indicate that adult female razorback suckers tagged with a passive integrated transponder (PIT) tags in Lake Mead were captured consistently at greater than 1-year intervals, while the majority of tagged, male fish were captured on an annual basis, thereby supporting historical ideas that female razorback suckers may exhibit non-annual spawning. Age at maturity for razorback sucker ranges from a minimum estimate of 2 years for male razorback sucker and 3 years for female razorback sucker to a maximum of 6 years for some populations, or occurs at sizes typically greater than 350 mm (Bestgen 1990).

Research efforts by Bozek et al. (1990) show that successful incubation of razorback sucker eggs in Lake Mohave occurs between 9.5 and 15.0°C, and in the laboratory successful embryo hatching occurs at 10-20°C. Hatching (eggs at a controlled 15°C) was reported to occur in 5.2-5.5 days (Minckley and Gustafson 1982). Egg mortality has been attributed to fluctuating water levels, scouring by currents and/or wave action, suffocation due to silt deposition, and nonnative egg predation (Minckley 1983, Bozek et al. 1984). Fertilized gametes are reported by Minckley and Gustafson (1982) as adhesive 3-4 hours post-fertilization, with cleavage being completed within 24 hours, gastrulation occurring at 34 hours, and blood circulation becoming established at 117 hours. Furthermore, all fins were reported to be fully formed and ossified at 64 days (27 mm TL) (Minckley and Gustafson 1982). Papoulias and Minckley (1990) found yolk absorption to occur approximately 8 days post-hatching (Minckley and Gustafson report 13 days at 15°C) and that the critical period during which exogenous feeding must occur to avoid mortality lies between 8-19 days after hatching. Papoulias and Minckley (1990) also found that the majority of larval mortality likely occurs within 20-30 days and is a result of starvation or receiving food too late after hatching, indicating that zooplankton levels are an important driver of larval razorback sucker survival (see diet section). Larval razorback sucker are photosensitive and display diel patterns in drift periodicity (Carter et al. 1986, Burke 1995).

Diet

Razorback sucker diet composition is highly dependant upon life stage, habitat, and food availability. Upon hatching, razorback sucker larvae have terminal mouths and shortened gut lengths (less than 1 body length) which in combination, appears to facilitate and necessitate selection of a wide variety of food types. Exogenous feeding occurs at approximately 10 mm TL (approximately 8-19 days), after which larvae from lentic systems feed mainly on phytoplankton and small zooplankton, while riverine larvae are assumed to feed largely on chironomids and other benthic insects (Minckley and Gustafeson 1982, Marsh and Langhorst 1988, Bestgen 1990, Papoulias and Minckley 1990, USFWS 1998). Papoulias and Minckley (1992) reared larval razorback sucker in three different ponds containing different densities of food resources to demonstrate that increased growth was positively related to invertebrate densities, suggesting the importance of larval food switching from algal and detrital food items to a diet enriched with invertebrates. Papoulias and Minckley (1990) show that larval razorback mortality is minimized when food levels are within the range of 50-1,000 organisms/L.

Later during growth (age and size information unknown, but at some point during the juvenile life stage), razorback sucker undergo an ontogenetic shift in mouth morphology, with the mouth becoming more inferior and allowing for more efficient access to benthic food sources. Thereafter, razorback sucker likely consume a variety of benthic-associated food items (USFWS 1998).

As adults, razorback sucker populations display unique diet compositions, depending upon whether the individual exists in a lacustrine or riverine setting (Bestgen 1990, USFWS 1998). Riverine fish consume a mixture of benthic invertebrates, algae, detritus, and inorganic materials, with little evidence of zooplankton consumption (Jonez and

Sumner 1954, Banks 1964, Vanicek 1967). Lacustrine-inhabiting adult razorback sucker consumption is dominated by cladoceran zooplankton, with some degree of algal and detrital material present in gut contents as well (Minckley 1973, Marsh 1987). While it is possible that razorback sucker may exhibit varying degrees of pelagic zooplanktivory, it is equally likely that the abundance of zooplankton noted in lentic-dwelling razorback sucker diets is simply the result of omnivorous benthic feeding. For example, Wurtsbaugh and Hawkins (1990) report large densities of zooplankton in samples collected from the profundal, hypolimnetic, water-substrate interface in Bear Lake, Utah-Idaho, particularly during daylight hours.

Age and Growth

Published growth estimates for razorback sucker vary, and available information is highly dependant upon life stage, habitat type, and overall ecological setting (Bestgen 1990, USFWS 1998). Information on growth is lacking for the early life stages of wild razorback sucker. The majority of growth information for larval and juvenile razorback sucker has been based largely on hatchery-produced fish (Brooks 1985, Marsh 1985, Marsh and Brooks 1989, Minckley et al. 1991, Mueller 1995). Razorback sucker that upon hatching are 7-9 mm can reach lengths of over 23 mm within 2 months (Papoulias and Minckley 1990). Subsequently, during the initial 6 years of life, young razorback sucker appear to grow rapidly (e.g., growth of 55-307 mm in 6 months for young razorback sucker stocked into ponds) (Osmundson and Kaeding 1989), after which growth becomes minimal (2 mm/year or less) as older age-classes are reached (McCarthy and Minckley 1987, Minckley et al. 1991). Studies of age and growth on the Lake Mohave razorback sucker population have shown that older adult fish show very little (approximately 2 mm per year or less), if any, growth (McCarthy and Minckley 1987). Modde et al. (1996) studied the largest extant riverine population of razorback sucker in the Green River, Utah, where he also found very slow growth in adults (1.66 mm per year). The highest growth rates described have been those reported for the Lake Mead razorback population, with growth rates of adult fish approaching 10-20 mm per year (outside of isolated refugia and hatchery ponds) (Holden 1999, Holden et al. 1997, 2000a, 2000b, 2001; Abate et al. 2002; Welker et al. 2003; Welker and Holden 2004). This population has been shown to be much younger overall than either the Lake Mohave or Green River populations, which likely accounts for the higher growth rates.

Past attempts to age razorback sucker using scales and other morphological structures were unsuccessful (McCarthy and Minckley 1987). The lack of clear annular marks, or irregular annuli that do not correspond to annuli found on other structures from the same fish, made aging razorback sucker reliably from scales problematic. This inability to accurately age individual fish using scales has also been a problem for other researchers working on wild razorback sucker populations in the Colorado River (McAda and Wydoski 1980, McCarthy and Minckley 1987) and on populations of white sucker (*Catostomus commersoni*) (Beamish 1973, Quinn and Ross 1982).

McCarthy and Minckley (1987) found pectoral fin rays to be a valid structure for use in aging young razorback sucker and used otoliths to determine that the razorback sucker population in Lake Mohave was 24-44 years of age in the 1980s. Beamish and Harvey

(1969) used the first four pectoral fin rays to age white sucker and found this method reliable. Quinn and Ross (1982) reported that pectoral fin rays were accurate in determining ages in younger (age 7 and under) populations of white sucker but that caution should be used in aging older and slower-growing fish.

During the early years of BIO-WEST razorback sucker studies on Lake Mead, two razorback sucker carcasses recovered from the lake were aged using both otoliths and pectoral fin rays to evaluate and develop a non-lethal technique for reliably aging razorback sucker populations in hopes of developing hypotheses pertaining to patterns of recruitment. While striving towards the development of a non-lethal aging technique, a dead, 381 mm TL razorback sucker of unknown sex was recovered from Echo Bay. Subsequently, another carcass was recovered from Las Vegas Bay (a 588 TL male). By using the combined carcasses, BIO-WEST was able to validate that in both fish, ages estimated from pectoral fin rays agreed with those obtained from sectioned otoliths. Both fish proved to be relatively young (ages 5 and 8) (Holden et al. 1999). Use of fin rays as a structure for aging has been further validated by aging multiple, known-age fish originating from Floyd Lamb State Park. Furthermore, attempts at aging razorback sucker from Lake Mead (more than 80 individual razorback suckers evaluated to date) demonstrates that ages ranged from 5 to 35+ years of age, with the majority of data being collected from wild, naturally recruited fish (Albrecht and Holden 2005).

Literature Cited

Abate, P.D., T.L. Welker, and P.B. Holden. 2002. Razorback sucker studies on Lake Mead, Nevada. 2001-2002 Annual Report. Prepared for the Department of Resources, Southern Nevada Water Authority, by BIO-WEST, Inc., Logan, Utah. PR-578-6.

Abbot, C.C. 1861. Descriptions of four new species of North American Cyprinidae. Proceedings of the Philadelphia Academy of Natural Sciences 12(1960):473-474.

Albrecht, B., and P.B. Holden. 2005. Razorback sucker studies on Lake Mead, Nevada. 2004-2005 Annual Report. Prepared for the Department of Resources, Southern Nevada Water Authority, by BIO-WEST, Inc., Logan, Utah. PR-960-1.

Banks, J.L. 1964. Fish species distribution in Dinosaur National Monument during 1961 and 1962.

Beamish, R.J. 1973. Determination of age and growth of populations of the white sucker (*Catostomus commersoni*) exhibiting a wide range in size at maturity. Journal of the Fisheries Research Board of Canada 30:607-616.

Beamish, R.J., and H.H. Harvey. 1969. Age determination in the white sucker. Journal of the Fisheries Research Board of Canada 26: 633-638.

Behnke, R.J., and D.E. Benson. 1980. Endangered and threatened fishes of the upper Colorado River Basin. Bulletin 503A, Cooperative Extension Service, Colorado State University, Fort Collins, Colorado.

Bestgen, K.R. 1990. Status review of the razorback sucker, *Xyrauchen texanus*, submitted to Bureau of Reclamation. Contribution 44, Larval Fish Laboratory, Colorado State University.

Bliesner, R., and V. Lamarra. 2005. Hydrology/Geomorphology/Habitat studies, 2004 annual report. San Juan River Basin Recovery Implementation Program, U.S. Fish and Wildlife Service, Albuquerque, New Mexico.

Boyd, C.E. 1979. Water quality in warm water fish ponds. Agricultural Experiment Station, Auburn University, Auburn, Alabama.

Bozek, M.A., L.J. Paulson, and J.E. Deacon. 1984. Factors affecting reproductive success of bonytail chubs and razorback suckers in Lake Mohave. Bureau of Reclamation, Final Report, Boulder City, Nevada.

Bozek, M.A., L.J. Paulson, and G.R. Wilde. 1990. Effects of ambient Lake Mojave temperatures on development, oxygen consumption, and hatching success of the razorback sucker. Environmental Biology of Fishes 27:255-263.

Bradford, R.H., S.D. Gurtin, and B.R. Vlach. 1998. Habitat use by razorback suckers implanted with ultra-sonic transmitters and released into the lower Imperial Division, Colorado River. Contract Report 03, Cooperative Agreement 3-FC-34-08243. Bureau of Reclamation and Arizona Game and Fish Department. Yuma, Arizona.

Bradford, R.H., and S.D. Gurtin. 2000. Habitat use by hatchery-reared adult razorback suckers released into the lower Colorado River, California-Arizona. North American Journal of Fisheries Management 20:154-167.

Brandenburg, W.H., M.A. Farrington, and S.J. Gottlieb. 2005. Colorado pikeminnow and razorback sucker larval fish survey in the San Juan River during 2004. San Juan River Basin Recovery Implementation Program, U.S. Fish and Wildlife Service, Albuquerque, New Mexico.

Bulkley, R.V., and R. Pimental. 1983. Temperature preference and avoidance by adult razorback sucker. Transactions of the American Fisheries Society 112:601-607.

Brooks, J.E. 1985. Annual reintroduction and monitoring report for razorback sucker *Xyrauchen texanus* in the Gila River basin, Arizona, 1985. U.S. Fish and Wildlife Service, Albuquerque, New Mexico.

Buth, D.G., R.W. Murphy, and L. Ulmer. 1987. Population differentiation and introgressive hybridization of the flannelmouth sucker and of hatchery and native stocks of the razorback sucker. Transactions of the American Fisheries Society 116:103-110.

Burke, T. 1995. Rearing wild razorback sucker larvae in lake-side backwaters, Lake Mohave, Arizona/Nevada. Proceeding of the Desert Fishes Council 26:35 (abstract only).

Carter, J.G., V.A. Lamarra, and R.J. Ryel. 1986. Drift of larval fishes in the upper Colorado River. Journal of Freshwater Ecology 3(4):567-577.

Douglas, M.E., and P.C. Marsh. 1998. Population and survival estimates of *Catostomus latipinnis* in Northern Grand Canyon, with distribution and abundance of hybrids with *Xyrauchen texanus*. Copeia 1998(4):915-925.

Douglas, P.A. 1952. Notes on the spawning of the humpback sucker, *Xyrauchen texanus* (Abbott). California Fish and Game 38(2):149-155.

Eastman, J.T. 1980. The caudal skeleton of catostomid fishes. American Midland Naturalist 103(1):133-148.

Ellis, M.M. 1914. Fishes of Colorado. University of Colorado Studies 11:1-136.

Gilbert, C.H., and N.B. Schofield. 1898. Notes on a collection of fishes from the Colorado Basin in Arizona. Proceedings of the U.S. National Museum 20:487-499.

Golden, M.E., and P.B. Holden. 2005. Retention, growth, and habitat use of stocked Colorado pikeminnow in the San Juan River: 2003-2004 annual report. San Juan River Basin Recovery Implementation Program, U.S. Fish and Wildlife Service, Albuquerque, New Mexico.

Gutermuth, F.B., L.D. Lentsch, and K.R. Bestgen. 1994. Collection of age-0 razorback suckers (*Xyrauchen texanus*) in the lower Green River, Utah. Southwestern Naturalist 39:389-390.

Holden, P.B. 1973. Distribution, abundance and life history of the fishes of the Upper Colorado River Basin. Ph.D. Dissertation. Utah State University, Logan.

Holden, P.B. (Editor). 1999. Flow recommendations for the San Juan River. San Juan River Basin Recovery Implementation Program, U.S. Fish and Wildlife Service, Albuquerque, New Mexico.

Holden, P.B., P.D. Abate, and J.B. Ruppert. 1997. Razorback sucker studies on Lake Mead, Nevada. 1996-1997 Annual Report. Prepared for the Department of Resources, Southern Nevada Water Authority, by BIO-WEST, Logan, Utah. PR-578-1.

Holden, P.B., P.D. Abate, and J.B. Ruppert. 1999. Razorback sucker studies on Lake Mead, Nevada. 1997-1998 Annual Report. Prepared for the Department of Resources, Southern Nevada Water Authority, by BIO-WEST, Logan, Utah. PR-578-2.

Holden, P.B., P.D. Abate, and J.B. Ruppert. 2000a. Razorback sucker studies on Lake Mead, Nevada. 1998-1999 Annual Report. Prepared for the Department of Resources, Southern Nevada Water Authority, by BIO-WEST, Logan, Utah. PR-578-3.

Holden, P.B., P.D. Abate, and J.B. Ruppert. 2000b. Razorback sucker studies on Lake Mead, Nevada. 1999-2000 Annual Report. Prepared for the Department of Resources, Southern Nevada Water Authority, by BIO-WEST, Logan, Utah. PR-578-4.

Holden, P.B., P.D. Abate, and T.L. Welker. 2001. Razorback sucker studies on Lake Mead, Nevada. 2000-2001 Annual Report. Prepared for the Department of Resources, Southern Nevada Water Authority, by BIO-WEST, Logan, Utah. PR-578-5.

Holden, P.B., and C.B. Stalnaker. 1975. Distribution of fishes in the Dolores and Yampa River systems of the upper Colorado Basin. Southwestern Naturalist 19:403-412.

Hubbs, C.L., L.C. Hubbs, and R.E. Johnson. 1943. Hybridization in nature between species of catostomid fishes. Contributions of the Laboratory of Vertebrate Biology, University of Michigan 22:1-76.

Hubbs, C.L., and R.R. Miller. 1953. Hybridization in nature between the fish genera *Catostomus* and *Xyrauchen*. Papers of the Michigan Academy of Sciences, Arts, and Letters 38:207-233.

Jackson, J.A. 2005. Nonnative control in the lower San Juan River 2004. San Juan River Basin Recovery Implementation Program, U.S. Fish and Wildlife Service, Albuquerque, New Mexico.

Johnson, J.E., and R.T. Hines. 1999. Effect of suspended sediment on vulnerability of young razorback suckers to predation. Transactions of the American Fisheries Society 128:648-655.

Jonez, A., and R.C. Sumner. 1954. Lakes Mead and Mojave investigations: A comparative study of an established reservoir as related to a newly created impoundment. Federal Aid to Fisheries Restoration Project Completion Report, F-1-R, Nevada Fish and Game Commission, Reno. 186 p.

Jordan, D.S. 1891. Report of explorations in Utah and Colorado during the summer of 1889, with an account of fishes found in each of the river basins examined. Bulletin of the U.S. Fish Commissioner 9:1-40.

Joseph, T.W., J.A. Sinning, R.J. Behnke and P.B. Holden. 1977. An evaluation of the status, life history, and habitat requirements of endangered and threatened fishes of the Upper Colorado River Basin. Report FWS/OBS-77/2, U.S. Fish and Wildlife Service, Fort Collins, Colorado.

Kirsch, P.H. 1889. Notes on a collection of fishes obtained in the Gila River at Fort Thomas, Arizona, by Lieut. W.L. Carpenter, U.S. Army. Proceedings of the U.S. National Museum 11:55-558.

Lanigan, S.H., and H.M. Tyus. 1989. Population size and status of the razorback sucker in the Green River basin, Utah and Colorado. North American Journal of Fisheries Management 9:68-73.

LaRivers, I. 1962. Fishes and fisheries of Nevada. Nevada Game and Fish Commission, Carson City.

Lee, J. 2005. Telemetry study update. Presentation at the 2nd Annual Lower Colorado River Native Fish Work Group Meeting. June 14, 2005, Laughlin, Nevada.

Marsh, P.C. 1985. Effect of incubation temperature on survival of embryos of native Colorado River fishes. Southwestern Naturalist 30:129-140

Marsh, P.C. 1987. Food of adult razorback sucker in Lake Mojave, Arizona-Nevada. Transactions of the American Fisheries Society 116:117-119.

Marsh, P.C. 1996. Threatened fishes of the world: *Xyrauchen texanus* (Abbott, 1860) (*Catostomidae*). Environmental Biology of Fishes 45:258.

Marsh, P.C., B.R. Kesner, and C.A. Pacey. 2005. Repatriation as a management strategy to conserve a critically imperiled fish species. North American Journal of Fisheries Management 25:547-556.

Marsh, P.C., C.A. Pacey, and B.R. Kesner. 2003. Decline of razorback sucker in Lake Mohave, Colorado River, Arizona and Nevada. Transactions of the American Fisheries Society 132:1251-1256.

Marsh, P.C., and J.E. Brooks. 1989. Predation by ictalurid catfishes as a deterrent to re-establishment of hatchery-reared razorback suckers. Southwestern Naturalist 34(2):188-195.

Marsh, P.C., and D.R. Langhorst. 1988. Feeding and fate of wild larval razorback sucker. Environmental Biology of Fishes 21:59-67.

Marsh, P.C., and W.L. Minckley. 1989. Observations on recruitment and ecology of razorback sucker: Lower Colorado River, Arizona-California-Nevada. Great Basin Naturalist 49(1):71-78.

McAda, C.W. 1977. Aspects of the life history of three catostomids native to the upper Colorado River basin. M.S. Thesis. Utah State University, Logan. 116 p.

McAda, C.W., and R.S. Wydoski. 1980. The razorback sucker, *Xyrauchen texanus*, in the upper Colorado River basin, 1974-76. U.S. Fish and Wildlife Service Technical Paper 99:1-15.

McCarthy, M.S., and W.L. Minckley. 1987. Age estimation for razorback sucker (*Pisces: Catostomidae*) from Lake Mohave, Arizona and Nevada. Journal of the Arizona-Nevada Academy of Sciences 21:87-97.

Medel-Ulmer, L. 1980. Movement and reproduction of the razorback sucker (*Xyrauchen texanus*) inhabiting Senator Wash Reservoir, Imperial County, California. Proceedings of the Desert Fishes Council 12(1980):106.

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

Minckley, W.L. 1983. Status of the razorback sucker, *Xyrauchen texanus* (Abbott), in the lower Colorado River basin. Southwestern Naturalist 28(2):165-187.

Minckley, W.L., and E.S. Gustafson. 1982. Early development of the razorback sucker, *Xyrauchen texanus* (Abbott). Great Basin Naturalist 42:55-561.

Minckley, W.L., P.C. Marsh, J.E. Brooks, J.E. Johnson, and B.L. Jensen. 1991. Management toward recovery of razorback sucker. Pages 303-357 *In:* W.L. Minckley and J.E. Deacon (editors). Battle against extinction: Native fish management in the American Southwest. University of Arizona Press. Tucson. 517 p.

Modde, T. 1996. Juvenile razorback sucker (*Xyrauchen texanus*) in a managed wetland adjacent to the Green River. Great Basin Naturalist 56:375-376.

Modde, T. 1997. Nursery suitability and razorback sucker use of a floodplain wetland in the middle Green River. Final Report submitted to the Recovery Implementation Program for the Recovery of Endangered Fishes in the Upper Colorado River Basin. U.S. Fish and Wildlife Service, Denver, Colorado.

Modde, T. 2005. Can habitat mitigate the impacts of nonnative species on rare native fishes: Observations from the Upper Colorado River Basin. Pages 123-128 *In:* M.J. Brouder, C.L. Springer and S.C. Leon, editors. Proceeding from two symposia: Restoring native fish to the lower Colorado River: Interactions of native and nonnative fishes, July 13-14, 1999, Las Vegas, Nevada; and Restoring natural function within a modified riverine environment: The lower Colorado River, July 8-9, 1998, Las Vegas, Nevada. U.S. Fish and Wildlife Service Southwest Region, Albuquerque, New Mexico.

Modde, T., Z.H. Bowen, and D.C. Kitcheyan. 2005. Spatial and temporal use of a spawning site in the Middle Green River by wild and hatchery-reared razorback suckers. Transactions of the American Fisheries Society 134:937-944.

Modde, T., K.P. Burnham, and E.J. Wick. 1996. Population status of the razorback sucker in the middle Green River (U.S.A.). Conservation Biology 10(1):110-119.

Modde, T., and D.G. Irving. 1998. Use of multiple spawning sites and seasonal movement by razorback sucker in the middle Green River, Utah. North American Journal of Fisheries Management 18:318-326.

Modde, T., R.T. Muth, and G. B. Haines. 2001. Floodplain wetland suitability, access and potential use by juvenile razorback suckers in the middle Green River, Utah. Transactions of the American Fisheries Society 130:1095-1105.

Moyle, P.B. 1976. Inland Fishes of California. University of California Press, Berkley.

Mueller, G.A. 1989. Observation of spawning razorback sucker (*Xyrauchen texanus*) utilizing riverine habitat in the lower Colorado River, Arizona-Nevada. Southwestern Naturalist 34:147-149.

Mueller, G. 1995. A program for maintaining the razorback sucker in Lake Mojave. American Fisheries Society Symposium 15:127-135.

Mueller, G.A., and P.C. Marsh. 1998. Post-stocking dispersal, habitat use, and behavioral acclimation of juvenile razorback suckers (*Xyrauchen texanus*) in two Colorado River reservoirs. U.S. Geological Survey Open File Report 98-301, Denver.

Mueller, G.A., P.C. Marsh, D. Foster, M. Ulibarri, and T. Burke. 2003. Factors influencing poststocking dispersal of razorback sucker. North American Journal of Fisheries Management 23(1):270-275.

Mueller, G.A., P.C. Marsh, G. Knowles, and T. Wolters. 2000. Distribution, movements, and habitat use of razorback sucker (*Xyrauchen texanus*) in a lower Colorado River reservoir, Arizona-Nevada. Western North American Naturalist 60(2):180-187.

Muth, R.T., G.B. Haines, S.M. Meismer, E.J. Wick, T.E. Chart, D.E. Snyder, J.M. Bundy. 1998. Reproduction and early life history of razorback sucker in the Green River, Utah and Colorado, 1992-1996. Draft Final Report for Project 34, submitted to the Recovery Implementation Program for the Endangered Fish Species in the Upper Colorado River Basin. Fort Collins, Colorado.

Osmundson, D.B. 2001. Flow regimes for restoration and maintenance of sufficient habitat to recover endangered razorback sucker and Colorado pikeminnow in the upper Colorado River: Interim recommendations for the palisade to riffle reach. Recovery Implementation Program Project No. 74. U.S. Fish and Wildlife Service, Final Report, Grand Junction, Colorado.

Osmundson, D.B., and L.R. Kaeding. 1989. Studies of Colorado squawfish and razorback sucker use of the 15-mile reach of the Upper Colorado River as part of conservation measures for the Green Mountain and Ruedi Reservoir water sales. U.S. Fish and Wildlife Service, Final Report, Grand Junction, Colorado.

Osmundson, D.B., and L.R. Kaeding. 1990. Recommendations for flows in the 15-mile reach during October-June for maintenance and enhancement of endangered fish populations in the upper Colorado River. Final Report, U.S. Fish and Wildlife Service, Grand Junction, Colorado.

Papoulias, D., and W.L. Minckley. 1990. Food limited survival of larval razorback sucker, *Xyrauchen texanus*, in the laboratory. Environmental Biology of Fishes 29:73-78.

Papoulias, D., and W.L. Minckley. 1992. Effects of food availability on survival and growth of larval razorback suckers in ponds. Transactions of the American Fisheries Society 121:340-355.

Piper, R.G., I.B. McElwain, L.E. Orme, J.P. McCraren, L.G. Fowler, and J.R. Leonard. 1982. Fish Hatchery Management. U.S. Fish and Wildlife Service, Department of the Interior, Washington, D.C.

Quinn, S.P., and M.R. Ross. 1982. Annulus formation by white suckers and the reliability of pectoral fin rays for ageing them. North American Journal of Fisheries Management 2:204-208.

Ryden, D.W. 2000. Monitoring of experimentally stocked razorback sucker in the San Juan River: March 1994-October 1997. Final Report, U.S. Fish and Wildlife Service, Colorado River Fishery Project, Grand Junction, Colorado.

Sigler, W.F., and R.R. Miller. 1963. Fishes of Utah. Utah State Department of Fish and Game, Salt Lake City, Utah.

Slaughter, J.E., S.D. Gurtin, J.A. Falke, S.J. Sampson, and R.H. Bradford. 2002. Habitat selection and use by hatchery reared adult razorback sucker and flathead catfish and the response of razorback sucker to off-channel habitat restoration activities within the Imperial Division, Lower Colorado River. Contract Report No. 5. Research Branch, Arizona Game and Fish Department.

Snyder, D.E., and R.T. Muth. 1990. Description and identification of razorback, flannelmouth, white, Utah, and mountain sucker larvae and early juveniles. Colorado Division of Wildlife, Denver. Technical Publication 38:1-152.

Snyder, D.E., R.T. Muth, and C.L. Bjork. 2004. Catostomid fish larvae and early juveniles of the upper Colorado River basin-morphological descriptions, comparisons, and computer-interactive key. Contribution 139 of the Larval Fish Laboratory, Colorado State University. Technical Publication No. 42, Colorado Division of Wildlife, Fort Collins, Colorado.

Taba, S.S., J.R. Murphy, and H.H. Frost. 1965. Notes on the fishes of the Colorado River near Moab, Utah. Proceedings of the Utah Academy of Science, Arts and Letters 42:280-283.

Tyus, H.M. 1987. Distribution, reproduction, and habitat use of the razorback sucker in the Green River, Utah, 1979-1986. Transactions of the American Fisheries Society 116:111-116.

Tyus, H.M., and C.A. Karp. 1989. Habitat use and streamflow needs of rare and endangered fishes, Yampa River, Colorado. U.S. Fish and Wildlife Service, Biological Reports 89(14):1-27.

Tyus, H.M., and C.A. Karp. 1990. Spawning and movements of the razorback sucker *Xyrauchen texanus* (Abbott) in the Green and Yampa river, Colorado and Utah. Southwestern Naturalist 35:427-433.

Tyus, H.M., C.W. Brown, and J.F. Saunders. 2000. Movements of young Colorado pikeminnow and razorback sucker in response to water flow and light level. Journal of Freshwater Ecology 15(4):525-535.

Tyus, H.M., and J.F. Saunders. 1996. Nonnative fishes in the upper Colorado River basin and a strategic plan for their control. Final Report to U.S. Fish and Wildlife Service 14-48-0006-95-923. Center for Limnology, University of Colorado, Boulder, Colorado.

[USFWS] U.S. Fish and Wildlife Service. 1998. Razorback sucker (*Xyrauchen texanus*) Recovery Plan. Denver, Colorado. 81 p.

[USFWS] U.S. Fish and Wildlife Service. 2002. Bonytail (*Gila elegans*) recovery goals: Amendment and supplement to the bonytail chub recovery plan. U.S. Fish and Wildlife Service, Mountain-Prairie Region 6, Denver Colorado.

Vanicek, C.D. 1967. Ecological studies of native Green River fishes below Flaming Gorge Dam, 1964-1966. Ph.D. Thesis, Utah State University, Logan. 125 p.

Welker, T.L. and P.B. Holden. 2003. Razorback sucker studies on Lake Mead, Nevada. 2002-2003 Annual Report. Prepared for the Department of Resources, Southern Nevada Water Authority, by BIO-WEST, Inc., Logan, Utah. PR-578-7.

Welker, T.L. and P.B. Holden. 2004. Razorback sucker studies on Lake Mead, Nevada. 2003-2004 Annual Report. Prepared for the Department of Resources, Southern Nevada Water Authority, by BIO-WEST, Inc., Logan, Utah. PR-578-8.

Wick, E.J., C.W. McAda, and R.V. Bulkley. 1982. Life history and prospects for recovery of the razorback sucker. Pages 120-126 *In:* W.H. Miller, H.M. Tyus, and C.A. Carlson (editors). Fishes of the upper Colorado River system: Present and future. American Fisheries Society, Bethesda, Maryland.

Wurtsbaugh, W.A., and C.P. Hawkins. (In Preparation). Tropic interactions between fish and invertebrates in Bear Lake, Utah-Idaho. Utah State University Ecology Center Final Report. Utah State University, Logan, Utah.

Wydoski, R. 2005. Update on netting efforts and razorback round-up, Colorado River downstream to the Lake Havasu Delta. Communicative message/update on the LCRFISH Listserver, February 24, 2005.

OTHER COVERED SPECIES

WESTERN RED BAT
(*Lasiurus blossevillii)*

Legal Status

The western red bat is not Federally listed as threatened or endangered. It was included in a draft list of Arizona Wildlife of Special Concern by the Arizona Game and Fish Department (AGFD in prep. in AGFD 2003). According to the State of Nevada Comprehensive Wildlife Conservation Strategy, the western red bat is a Nevada Species of Conservation Priority and is protected and considered sensitive (Nevada Department of Wildlife 2005). In California, the western red bat is proposed as a Mammal of Special Concern (Bolster 2005). The U.S. Forest Service considers it a sensitive animal in region 3 (AGFD 2003). The Western Bat Working Group (1998) lists the western red bat as a species of "Red or High" priority, the highest priority available.

Distribution

Historical and Current Range

The historical range of the western red bat is believed to mirror its current range, where available habitat occurs, including the western portions of British Columbia, Washington and Oregon, and western, central, and southern Nevada. The entire states of California and Arizona are considered within its range. One range map contains all of Utah, Colorado, Wyoming, Idaho, Montana, New Mexico, and most of western Texas (Natureserve 2006). Kays and Wilson (2002) restrict the species' range in Utah to the west, in New Mexico to the southwest, and include only the extreme western portion of Texas. Western Texas is also an area of overlap between western and eastern red bats (*Lasiurus borealis*), which occur in the central and eastern areas of the United States (Genoways and Baker 1988, Kays and Wilson 2002). Western red bat distribution continues throughout Mexico, Central America, and almost the entire continent of South America (Natureserve 2006).

Populations within LCR MSCP Project Boundary

Western red bat demographics within the LCR MSCP project boundary are not well known. Historically, no red bats had been reported from the LCR. The closest record to the LCR was of 3 female red bats collected in July 1902 on Big Sandy Creek (50 miles east of Topock, AZ) (Hoffmeister 1986). While using mist-nets, harp traps, and acoustic sampling to study bats along the Muddy River in Moapa Valley in Clark County, Nevada, Williams (2001) found that red bats were the sixth-most abundant species acoustically detected. Western red bats have also been recorded acoustically from March through October along the Las Vegas Wash, which drains all runoff from Las Vegas, Nevada, into Lake Mead. Red bats have been detected during migration and males and/or non-reproductive females have been detected during the summer months (O'Farrell 2006). In January 2002, a male red bat was mist-netted on the Bill Williams National Wildlife

Refuge (BWRNWR) approximately 7 miles from Lake Havasu (Brown 2006). During a survey from 2001 to 2002, red bats were recorded acoustically at the BWRNWR, Havasu National Wildlife Refuge (HNWR), and the Imperial National Wildlife Refuge (INWR), although they only accounted for 0.14% of the total call minutes (Brown 2006). The HNWR is located along approximately 30 miles of the Colorado River from Needles, California, to Lake Havasu City, Arizona. The INWR is located along about 30 miles of the Colorado River 38 miles north of Yuma, Arizona. The western red bat appears to be rare along most of the LCR, but continued surveys may find more populations.

Life History

General Description

The western red bat was previously recognized as a subspecies of the eastern red bat (*Lasiurus borealis teliotis*) (Cockrum 1960). It was acknowledged as a separate species by most bat researchers between 1988 and 1995 (Baker et al. 1988, Morales and Bickham 1995).

The western red bat is a medium-sized bat with pelage that is usually mottled reddish and grayish, but can range from bright orange to yellow-brown. Whitish patches can be seen near the shoulder and most pelage hairs are frosted with white tips. Wings are long, narrow, and pointed. They have a distinct bib under the neck, which contrasts greatly with the jet-black wing membrane. Ears are 11-13 mm in length, low and rounded, and the tragus is short and blunt. Males are usually more colorful than females. Forearm measures 3.8-4.3 cm, weight is 7-15 g, and wingspan is 29.0-33.2 cm. The western red bat differs only slightly with the eastern red bat, which is generally larger and has more frosted hairs than *L. blossevillii*. When possible, it is best to distinguish the two species by range. Western red bats can be distinguished from other bats not in the genus' *Lasiurus* and *Lasionycteris* in Arizona by their short ears and long tail membrane. Silver-haired bats (*Lasionycteris noctivagans*) have black hairs with silver tips while red bats do not. Two other *Lasiurus* species that have overlapping distributions with *L. blossevillii* are *L. cinereus* (hoary bat) and *L. xanthinus* (western yellow bat). Hoary bats are larger (forearm 5.0-5.4 cm) and have black fur around the edges of the ears. Western yellow bats are also larger (forearm 4.5-5.0 cm), generally yellowish in color and only the anterior half of the uropatagium is furred (Kays and Wilson 2002, AGFD 2003).

Western red bats are mostly solitary, but may migrate in groups and forage in close association with others. Males and females migrate at different times and have different summer ranges. They normally migrate south in the winter and may be active in areas with temperatures as low as 12º-18ºC (55º-65ºF).

Breeding

Breeding occurs from August to October and the female will store sperm until the following spring when fertilization begins. After a gestation of 60-70 days, a female gives birth to a litter of two offspring (average is 2.3) from late May to mid-June. Estimated time of young to fledge is between their third and fourth week (AGFD 2003).

Diet and Foraging

Western red bats begin foraging 1-2 hours after dark and may continue into the following morning. They are known to feed 600-1000 yards from their roosting site and will forage from tree top level to a few feet above the ground. Moths appear to be one of the main prey items but the bats also readily feed on flies, bugs, beetles, cicadas, ground-dwelling crickets, and hymenopterans. They use their wing membranes to capture prey and will sometimes land on vegetation to catch an insect. Red bats commonly forage near light sources, which attract insects. Echolocation is used to find prey, including narrow and broadband calls. When searching, they use long calls with a low pulse repetition of narrow band frequencies. They will fix on a target about 5-10 m away and, on average, will attack prey every 30 seconds. Red bats are found to be successful 40% of the time (AGFD 2003).

Habitat

Like other members of the genus *Lasiurus*, western red bats primarily roost in trees (Shump & Shump 1982, Cryan 2003, and AGFD 2003). Specifically, in the southwestern United States, *L. blossevillii* is found in desert riparian areas. Tree species in these areas may include but are not limited to Fremont cottonwood (*Populus fremontii*), Goodding's willow (*Salix gooddingii*), and Sycamore (*Platanus* spp.). Although red bats may roost in any of these, they primarily roost in cottonwoods. They are also known to roost in shrubs in riparian habitats, as well as fruit tree orchards (AGFD 2003). If roosting in dense foliage, they can resemble dead leaves (AGFD 2003). Red bats have been observed occasionally roosting in cave-like situations and in the boot of the saguaro cactus, a hardened area of scar tissue that forms a hollow shape inside the cactus from cavity excavation by woodpeckers (E.L. Cockrum pers. comm. 1992 in AGFD 2003). Palo Verde (*Cercidium floridum*) trees have been used as roost sites for the sympatric hoary bat (Brown 2006).

Elevation of these habitats may vary from 580 to 2,196 m (1,900 to 7,200 ft). Roost sites range from a few feet off the ground to more than 40 ft high. Red bats prefer heavily shaded areas, which are open underneath, enabling them to drop into flight. Williams (2001) found that western red bats will use a variety of habitat in a riparian area. He collected capture and acoustic data along the Muddy River in Moapa Valley, Nevada, for 15 different bat species. Four habitat types were distinguished: riparian marsh, mesquite bosque, riparian woodland (either broadleaf trees or exotic palm trees), and riparian shrubland, which consists of arrowweed (*Pluchea sericea*) and quailbush (*Atriplex lentiformis*). Williams found that western red bats used all 4 habitat types, with the riparian marsh being the least utilized.

Threats

Predators known to be threats to red bats include birds of prey, roadrunners (*Geococcyx californianus*), opossums (*Didelphis virginiana*), and domestic cats (Shump and Shump 1982). Specifically, short-eared owl (*Asio flammeus*) pellets in San Diego were found to have parts of a western red bat (Huey 1926). Woodpeckers and raccoons have been observed disturbing other tree-roosting bat species at their roosting sites (Sparks et al. 2003). The greatest threat to western red bats in the southwestern United States is the loss of riparian habitat. Specifically, the Western Bat Working group has stated that the loss of cottonwood forests from desert riparian corridors may be the reason for the decline of the western red bat in those areas. These forests may be important to not only resident red bats, but also to migrants (AGFD 2003). Eastern red bats have been observed hibernation-roosting in leaf litter on the forest floor, which can be a threat in the event of a fire (Moorman et al. 1999).

Human-caused threats include barbwire fences and motor vehicles (Baker 1983, in Myers and Hatchett 2000). Pesticide use in fruit orchards may also pose a threat to bats roosting at those sites (Bolster 2005). The negative image of bats by the public, such as the fear of rabies, should also be considered a threat (Fenton 1997).

LCR MSCP Conservation Measures

WRBA1 — Conduct surveys to determine the distribution of the western red bat.
Conduct investigations to identify the distribution of the western red bat in reaches 3-5.

WRBA — Create 765 acres of western red bat roosting habitat.
Of the 7,260 acres of cottonwood-willow and honey mesquite to be created as covered species habitat, at least 765 acres will be designed and created to provide western red bat roosting habitat. Created roosting habitat will be designed and managed to support cottonwood-willow types I and II and honey mesquite type III. The LCR MSCP process for selecting sites to establish cottonwood-willow and honey mesquite as habitat for other covered species habitat will, based on the information collected under conservation measure WRBA1, give priority, when consistent with achieving LCR MSCP goals for other covered species, to selecting sites that are occupied by the western red bat in reaches 3-5. Created cottonwood-willow and honey mesquite land cover will be designed to establish stands that will support a substantially greater density and diversity of plant species that will provide roost trees and that are likely to support a greater abundance of insect prey species than is currently produced in the affected land cover types (LCR MSCP 2004).

LCR MSCP Research and Monitoring Needs

The two main factors that affect bat habitat use are roost selection/availability and prey abundance (Fenton 1997). The Western Bat Working Group has acknowledged that information is lacking for the western red bat (Bolster 2005).

To determine roost selection criteria along the LCR, western red bats can be studied using radio telemetry techniques. Brown (1996) previously tracked hoary bats in a telemetry study along the Bill Williams River, and was able to locate their roosting sites in cottonwood and willow trees. Brown has suggested that because hoary bats are slightly more common than red bats, they could be used as an indicator species for habitat creation projects. In the Pacific Northwest, Campbell et al. (1996) found differences between trees used as roosting sites and trees chosen randomly in a study of silver-haired bats. Radio telemetry could be used for studying western red bat behavior; however, obtaining a sufficient sample size may be difficult because they are difficult to capture and they are rarely found together in large numbers (Williams 2001).

Habitat patch size may affect roost selection and foraging. Since western red bats do not exhibit colonial behavior, large populations are not available to study population trends (Carter et al. 2003). Habitat use trends can be monitored to ensure the habitat continues to supply adequate foraging and roosting characteristics over a long period.

Seasonal movements and habitat use are currently unknown for red bats along the LCR. Telemetry may also be useful in determining migration patterns or location and characteristics of wintering habitat. Hoary bats may be used as a surrogate species to determine how tree roosting bats use restored riparian habitat.

Prey abundance and foraging within habitat mosaics may be an important limiting factor for red bats. Bats will forage wherever they can locate prey. Williams (2001) found red bats using riparian shrublands and mesquite bosques to forage. It may be beneficial to conduct acoustic surveys adjacent to restoration sites to determine how much other habitat in the area (i.e., agricultural fields, saltcedar stands) is being used as foraging habitat. Pesticide use adjacent to sites is also important to investigate as prey abundance and the health of the bats themselves may be affected.

Other Potential Research and Monitoring Opportunities

Currently, the only large western red bat populations known near the LCR MSCP planning area are in Moapa Valley and possibly at Las Vegas Wash. Mist-netting should be performed at Las Vegas Wash to determine if the individuals using the area during the summer are breeding.

Literature Cited

Arizona Game and Fish Department. 2003. *Lasiurus blossevillii*. Unpublished abstract compiled and edited by the Heritage Data Management System, Arizona Game and Fish Department, Phoenix, Arizona. 7 p. http://www.gf.state.az.us/w_c/edits/documents/Lasiblos.fi.pdf. Accessed January 6, 2006.

Baker, R.J., J.C. Patton, H.H. Genoways, and J.W. Bickham. 1988. Genic studies of *Lasiurus* (Chiroptera: Vespertilionidae). Occasional Papers The Museum Texas Tech University 117:1-15.

Bolster, B.C. 2005. *Lasiurus blossevillii* western red bat. Western Bat Working Group (webpage). http://wbwg.org/species_accounts.htm#LABL. Accessed January 6, 2006.

Brown, P.E. 1996. Bat survey of the Bill Williams River. Arizona Game and Fish Department Heritage Fund Project I93073, administered by Bureau of Land Management Lake Havasu, Arizona.

Brown, P. 2006. Lower Colorado River bat monitoring protocol. Draft report submitted to USBR. 30 p.

Burles, D.W., and R.A. Ring. 2005. Which colour of light should you use to monitor insect abundance? Western Bat Working Group Abstracts from the 2005 Biennial Meeting (webpage). http://www.wbwg.org/abstract.htm. Accessed January 6, 2006.

Campbell, L.A., J.G. Hallett, and M.A. O'Connell. 1996. Conservation of bats in managed forests: Use of roosts by *Lasionycteris noctivagans*. Journal of Mammalogy 77:976-984.

Carter, T.C., M.A. Menzel, D.A. Saugey. 2003. Population trends of solitary foliage-roosting bats. Pages 41-47 *In:* O'Shea, T.J. and M.A. Bogan (editors). Monitoring trends in bat populations of the United States and territories: Problems and prospects: U. S. Geologic Survey, Biological Resources Discipline, Information and Technology Report, USGS/BRD/ITR—2003-003, 274 p.

Cockrum, E.L. 1960. The recent mammals of Arizona: Their taxonomy and distribution. The University of Arizona Press, Tucson. p. 53-54.

Cryan, P.M. 2003. Seasonal distribution of migratory tree bats (*Lasiurus* and *Lasionyteris*) in North America. Journal of Mammalogy 84:579-593.

Fenton, M.B. 1997. Science and the conservation of bats. Journal of Mammalogy 78:1-14.

Genoways, H.H. and R.J. Baker. 1988. *Lasiurus blossevillii* (Chiroptera: Vespertilionidae) in Texas. The Texas Journal of Science 40:111-113.

Hoffmeister, D.F. 1986. Mammals of Arizona. University of Arizona Press. Tucson. p. 97-100.

Huey, L.M. 1926. General notes: Bats eaten by short-eared owl. Auk 43:96-97.

Kays, R.W., and D.E. Wilson. 2002. Mammals of North America. Princeton University Press. Princeton, NJ. p. 144.

Lower Colorado River Multi-Species Conservation Program. 2004. Lower Colorado River Multi-Species Conservation Program, Volume II: Habitat Conservation Plan. p. 5-52.

Moorman, C.E., K.R. Russell, M.A. Menzel, S.M. Lohr, J.E. Ellenberger, and D.H. Van Lear. 1999. Bats roosting in deciduous leaf litter. Bat Research News 40:74-75.

Morales, J.C., and J.W. Bickham. 1995. Molecular systematics of the genus *Lasiurus* (Chiroptera: Vespertilionidae) based on restriction-site maps of the mitochondrial ribosomal genes. Journal of Mammalogy 76:730-749.

Myers, P., and J. Hatchett. 2000. *Lasiurus borealis* (red bat). Animal Diversity Web (webpage). http://animaldiversity.ummz.umich.edu/site/accounts/information/Lasiurus_borealis.html. Accessed April 4, 2006.

NatureServe. 2006. Comprehensive report species – *Lasiurus blossevillii*. NatureServe Explorer (webpage). http://www.natureserve.org/explorer/servlet/NatureServe?searchName=Lasiurus+blossevillii. Accessed March 22, 2006.

Nevada Department of Wildlife. 2005. Identification of species of conservation priority. State of Nevada Comprehensive Wildlife Conservation Strategy. p. 59.

O’Farrell Biological Consulting. 2006. Final report: Long-term acoustic monitoring of bat populations associated with an extensive riparian restoration program in Las Vegas Wash, Clark County, Nevada (2004-2005). Prepared for Southern Nevada Water Authority.

Shump, K.A. Jr., and A.U. Shump. 1982. *Lasiurus borealis*. Mammalian Species 183:1-6.

Sparks, D.W., M.T. Simmons, C.L. Gummer, and J.E. Duchamp. 2003. Disturbance of roosting bats by woodpeckers and raccoons. Northeastern Naturalist 10:105-108.

Western Bat Working Group. 1998. Regional bat species priority matrix. Western Bat Working Group (webpage). http://www.wbwg.org/spp_matrix.html. Accessed April 6, 2006.

Williams, J.A. 2001. Community structure and habitat use by bats in the upper Moapa Valley, Clark County, Nevada. Unpublished M.A.S. Thesis. University of Nevada, Las Vegas. p. 55.

WESTERN YELLOW BAT
(*Lasiurus xanthinus*)

Legal Status

The western yellow bat is not Federally listed as threatened or endangered. It was included in a draft list of Arizona Wildlife of Special Concern by the Arizona Game and Fish Department (AGFD 2003). According to the State of Nevada Comprehensive Wildlife Conservation Strategy, the western yellow bat is a Nevada Species of Conservation Priority (Nevada Department of Wildlife 2005). California Department of Fish and Game (CDFG) have proposed it as a species of special concern (B. Bolster personal communication). The Western Bat Working Group (1998) lists the western yellow bat as a species of “Red or High” priority, the highest priority available.

Distribution

Historical Range

The first known occurrence of the western yellow bat in the United States was found in Palm Springs, California, in November 1945 (Constantine 1946). No other individuals were observed in the United States until January and February 1960, when two yellow bats were found roosting in dead palm fronds while trees were being trimmed at the University of Arizona in Tucson (Cockrum 1961). In 1963, yellow bats were captured in Guadalupe Canyon, New Mexico, along a riparian corridor (Mumford & Zimmerman 1963). Constantine (1966) also published the finding of yellow bats in Scottsdale, Phoenix, and Yuma, Arizona. The yellow bat’s historic range appears to be southern New Mexico, west through central Arizona and southern California, and southward into central and western Mexico, including Baja California (Kays & Wilson 2002).

Current Range

General range maps for the western yellow bat include the southern portion of California, the southern half of Arizona, and the southwestern corner of New Mexico. The range continues south into Baja, California, and west and central Mexico (Kays & Wilson 2002 and NatureServe 2006). The species has recently been found as far north as Moapa Valley in southern Nevada (O’Farrell et al. 2004). Currently, it is believed that the western yellow bat has expanded its range across the southwestern United States northward with the introduction of the Washington fan palm (*Washingtonia robusta*) (AGFD 2003). It has also been confirmed farther east, in Big Bend National Park in Texas (Higginbotham et al. 1999).

Populations within LCR MSCP Project Boundary

Western yellow bat populations are not well known within the Lower Colorado River Multi-Species Conservation Program (LCR MSCP) boundary. The first known occurrence along the lower Colorado River (LCR) was in Yuma, Arizona (Constantine

1966). In 1980, a yellow bat was turned in for rabies testing in Blythe, California (Constantine 1998). During a survey along the Bill Williams River in 1996, three western yellow bats were captured near Planet Ranch, one of which was later found in Lake Havasu City with the aid of a radio transmitter (Brown 1996). In an LCR-wide survey conducted from 2001 to 2002, they were picked up acoustically at the Bill Williams River National Wildlife Refuge (NWR), Imperial NWR, and north of Parker, Arizona (Brown 2006). Western yellow bats have been detected acoustically during all months, except for January, at Las Vegas Wash (O'Farrell 2006). A year-round resident breeding population was found in Moapa Valley, Nevada, near the Muddy River, a tributary of the Colorado River (O'Farrell et al. 2004). Yellow bats were the second-most abundant bat species detected in Moapa Valley (Williams 2001). The Moapa Valley population is the largest known breeding population near the LCR MSCP boundary.

Life History

General Description

The western yellow bat was previously recognized as a subspecies of the southern yellow bat (*Lasiurus ega xanthinus*) (Mumford and Zimmerman 1963). They were separated into two distinct species between 1988 and 1995 (Baker et al. 1988, Morales and Bickham 1995). Some continued to recognize *L. e. xanthinus* through 1995 (Kurta and Lehr 1995).

The western yellow bat is a medium to large-sized bat, whose pelage is yellowish-buff to light brownish, with fur tipped with gray or white. Forearm ranges from 41.5 to 49.0 mm, weight averages 9.2 to 22.5 g, and wingspan ranges from 33.5 to 35.5 cm. Ears are shorter than many other species, but their length is larger than their width (17.0 mm long). The anterior half of dorsal surface of the uropatagium is well-furred, while the posterior half is bare or almost bare (AGFD 2003). The western yellow bat differs from the southern yellow bat by having a brighter yellow pelage, especially on the tail membrane; however, this characteristic is difficult to detect so it is best to distinguish the two by range (Kays and Wilson 2002). In Arizona, western yellow bats can be distinguished from other bats not in the genera *Lasiurus* and *Lasionycteris* by their short, round ears and long tail membrane, with at least the anterior portion well-furred. *Lasionycteris* spp. hair is black with silver tips while yellow bats never have black hairs. Uropatagium is completely furred in other *Lasiurus* spp. found in Arizona. Hoary bats (*Lasiurus cinereus*) are larger (forearm 50.0-57.0 mm), have black edging around the ears, and have a mahogany brown pelage that is distinctly silver tipped. The western red bat (*Lasiurus blossevillii*) is smaller (forearm 38.0-43.0 mm) and has a reddish pelage. Also, yellow bats are known to be sexually dimorphic in size, with females being slightly larger (forearm 2 mm larger) (AGFD 2003).

Western yellow bats are solitary and were thought to not migrate (AGFD 2003). However, Williams (2001) found that populations in southern Nevada decline in the winter months and almost every individual captured during this time was male. It is unknown if this is a long-distance migration, local migration, or habitat shift by females in the winter. In Arizona, Cockrum (pers. comm. in AGFD 2003) found that males were generally found in the spring and summer and females were found from midwinter to mid-spring. Females usually give birth to two young in early June-July, and pregnant females have been found as early as late April (AGFD 2003, Kurta and Lehr 2005). Breeding biology is not well understood. Breeding time is unknown; however, it is thought that females store sperm and both males and females probably can breed within their first year (Kurta & Lehr 1995). It is unknown if they breed in Arizona, but reproductive females were common in southern Nevada (AGFD 2003 and Williams 2001).

Diet

Western yellow bats feed on a variety of insects including Hymentoptera, Diptera, Lepidoptera, Coleoptera, Hemiptera, and Orthoptera (Higginbotham et al. 1999, O'Farrell et al. 2004). They are known to leave day roosts and begin foraging at dusk. Yellow bats have been captured over water holes but it is unknown if they were foraging or drinking (Mumford and Zimmerman 1963). Williams (2001) found that yellow bats in Moapa Valley, Nevada, were more active acoustically in riparian woodlands, rather than other habitats in the area. There is also evidence that they forage in this habitat type more than in the others.

Habitat

Western yellow bats are known to roost in the dead palm frond skirts of fan palms (*Washingtonia* spp.) (Cockrum 1961, Kurta and Lehr 1995, Williams 2001). In Guadalupe Canyon, New Mexico, broadleaf deciduous riparian trees, such as Fremont cottonwood (*Populus fremontii*), sycamore (*Platanus wrightii*), and hackberry (*Celtis reticulate*), were used as roosting sites (Mumford and Zimmerman 1963). In the Big Bend region of Texas, a western yellow bat was found using the giant dagger yucca (*Yucca carnerosana*) as a roosting site, in a similar manner as those using palm trees (Higginbotham et al. 2000). Roosting height can range from 2.2 m from the ground to the tallest palm or deciduous trees in the area (Higginbotham et al. 2000, Mumford and Zimmerman 1963). Palm trees may be preferred because dead fronds closely match their fur coloration, although they will utilize any tree that gives them enough cover to be hidden while roosting. In Arizona, they are found at elevations from 168 to 1,830 meters (AGFD 2003). Along the LCR, yellow bats have been recorded at a cottonwood revegetation site at Imperial NWR and a dense palm grove just north of Parker, Arizona (Brown 2006).

Known predators of the western yellow bat include domestic cats and dogs, and barn owls (Kurta and Lehr 1995). Predators of other foliage roosting bats include birds of prey, roadrunners, and opossums (Shump & Shump 1982). Woodpeckers and raccoons have been observed disturbing other tree-roosting species at their roosting sites (Sparks et al. 2003). Human threats include barbwire fences and vehicles, as well as the negative image many have about bats (Baker 1983, in Myers & Hatchett 2000 and Fenton 1997). The use of pesticide threatens both bats and their insect prey. The cosmetic trimming of palm trees is probably one of the primary threats to yellow bats. The major threat to most bat species is the loss of habitat, including open water, which degrades roosting and foraging areas (Williams 2005).

LCR MSCP Conservation Measures

WYBA1 – Conduct surveys to determine the distribution of the western yellow bat.
Conduct investigations to identify the distribution of the western yellow bat in reaches 3-5.

WYBA2 – Avoid removal of western yellow bat roost trees.
To the extent practicable, avoid removal of palm trees that could serve as roosts for the western yellow bat when creating covered species habitats.

WYBA3 – Create 765 acres of western yellow bat roosting habitat.
Of the 7,260 acres of cottonwood-willow and honey mesquite to be created as covered species habitat, at least 765 acres will be designed and created to provide western yellow bat roosting habitat. Created roosting habitat will be designed and managed to support cottonwood-willow types I and II and honey mesquite type III. The LCR MSCP process for selecting sites to establish cottonwood-willow and honey mesquite as habitat for other covered species habitat will, based on the information collected under conservation measure WYBA1, give priority, when consistent with achieving LCR MSCP goals for other covered species, to selecting sites that are occupied by the western yellow bat in reaches 3-5. Created cottonwood-willow and honey mesquite land cover will be designed to establish stands that will support a substantially greater density and diversity of plant species that will provide roost trees and that are likely to support a greater abundance of insect prey species than is currently produced in the affected land cover types (LCR MSCP 2004).

LCR MSCP Research and Monitoring Needs

The two main factors that affect bat habitat use are roost selection/availability and prey abundance (Fenton 1997). The Western Bat Working Group has listed five data deficiencies for western yellow bats, including distribution, migration, habitat requirements, activity patterns (both daily and seasonally), and threats, including palm frond trimming and pesticide use (Bolster 2005).

Radio telemetry studies can be used to obtain data on western yellow bats. Brown (1996) placed transmitters on three individuals on the Bill Williams River, near Planet Ranch. Roosting sites were discovered for two of the bats and distances between roost and capture sites were measured. Because yellow bats appear to favor palm trees as roosting sites, telemetry could be used to determine yellow bat habitat selection in created cottonwood and willow habitats. In the Pacific Northwest, Campbell et al. (1996) found differences between trees used by silver-haired bats as roosting sites and trees chosen randomly. Similar research techniques could be used to better understand roost selection and other habitat requirements, including minimum patch size, for yellow bats.

Acoustic techniques can be used to determine seasonal habitat use and migration patterns for yellow bats. Prey abundance and foraging, with respect to habitat type, may be important limiting factors for yellow bats. Bats will forage wherever they can locate prey. Williams (2001) found yellow bat activity concentrated in riparian woodlands, with some activity observed in riparian shrublands and other habitats in the area. It may be beneficial to conduct acoustic surveys adjacent to restoration sites to determine how much other habitat in the area (i.e., agricultural fields, saltcedar stands) is being used as foraging habitat. Pesticide use adjacent to sites may also be important due to lower prey abundance and potential negative health effects on the bats themselves. Prey abundance studies can be performed, using light traps, in specific habitat types to monitor nocturnal insects, including species diversity and abundance. It should be noted that it may be necessary to use a variety of different colored lights in the traps to gain the highest diversity of insects found at a site (Burles and Ring 2005).

Brown (2006) has suggested using hoary bats as a surrogate species when monitoring riparian habitat creation projects because they were historically detected along the LCR and are more common than yellow bats. Because Lasiurine bats do not exhibit colonial behavior, they cannot be studied for population trends (Carter et al. 2003). Monitoring habitat availability during the course of the 50-year LCR MSCP will offer data that can be used to determine whether created habitat will continue to supply yellow bats with adequate sites for roosting and foraging over a long period of time.

Literature Cited

Arizona Game and Fish Department. 2003. *Lasiurus xanthinus*. Unpublished abstract compiled And edited by the Heritage Data Management System, Arizona Game and Fish Department, Phoenix, AZ. 6 p. http://www.gf.state.az.us/w_c/edits/documents/Lasixant.fi.pdf. Accessed January 6, 2006.

Baker, R.J., J.C. Patton, H.H. Genoways, and J.W. Bickham. 1988 Genic studies of *Lasiurus* (Chiroptera: Vespertilionidae). Occasional Papers The Museum Texas Tech University 117:1-15.

Bolster, B.C. (update by J.A. Williams). 2005. *Lasiurus xanthinus* western yellow bat. Western Bat Working Group (webpage) http://www.wbwg.org/species_accounts.htm#LAXA. Accessed January 6, 2006.

Brown, P.E. 1996. Bat survey of the Bill Williams River. Arizona Game and Fish Department Heritage Fund Project I93073, administered by Bureau of Land Management Lake Havasu, Arizona.

Brown, P. 2006. Lower Colorado River bat monitoring protocol. Draft report submitted to USBR. 30 p.

Burles, D.W., and R.A. Ring. 2005. Which colour of light should you use to monitor insect abundance? Western Bat Working Group Abstracts From the 2005 Biennial Meeting (webpage). http://www.wbwg.org/abstract.htm. Accessed January 6, 2006.

Campbell, L.A., J.G. Hallett, and M.A. O'Connell. 1996. Conservation of bats in managed forests: Use of roosts by *Lasionycteris noctivagans*. Journal of Mammalogy 77:976-984.

Carter, T.C., M.A. Menzel, D.A. Saugey. 2003. Population trends of solitary foliage-roosting bats. Pages 41-47 *In:* O'Shea, T.J. and M.A. Bogan, (editors). Monitoring trends in bat populations of the United States and territories: Problems and prospects: U.S. Geologic Survey, Biological Resources Discipline, Information and Technology Report, USGS/BRD/ITR—2003-003, 274 p.

Cockrum, E.L. 1961. Southern yellow bat in Arizona. Journal of Mammalogy 42:97.

Constantine, D.G. 1946. A record of *Dasypterus ega xanthinus* from Palm Springs, California. Bulletin of the Southern California Academy of Science 45:107.

Constantine, D.G. 1966. New bat locality records from Oaxaca, Arizona, and Colorado. Journal of Mammalogy 47:125-126.

Constantine, D.G. 1998. Range extensions of ten bats in California. Bulletin of the Southern California Academy of Science 97:49-75.

Fenton, M.B. 1997. Science and the conservation of bats. Journal of Mammalogy 78:1-14.

Higginbotham, J.L., M.T. Dixon, and L.K. Ammerman. 2000. Yucca provides roost for *Lasiurus xanthinus* (Chiroptera: Vespertilionidae) in Texas. The Southwestern Naturalist 45:338-340.

Kays, R.W., and D.E. Wilson. 2002. Mammals of North America. Princeton University Press, Princeton, NJ. p. 144.

Kurta, A., and G.C. Lehr. 1995. *Lasiurus ega*. Mammalian Species 515:1-7.

Lower Colorado River Multi-Species Conservation Program. 2004. Lower Colorado River Multi-Species Conservation Program, Volume II: Habitat Conservation Plan. p. 5-53 through 5-54.

Morales, J.C., and J.W. Bickham. 1995. Molecular systematics of the genus *Lasiurus* (Chiroptera: Vespertilionidae) based on restriction-site maps of the mitochondrial ribosomal genes. Journal of Mammalogy 76:730-749.

Mumford, R.E., and D.A. Zimmerman. 1963. The southern yellow bat in New Mexico. Journal of Mammalogy 44:417-418.

Myers, P., and J. Hatchett. 2000. *Lasiurus borealis* (red bat). Animal Diversity Web (webpage). http://animaldiversity.ummz.umich.edu/site/accounts/information/Lasiurus_borealis.html. Accessed April 4, 2006.

NatureServe. 2005. *Lasiurus xanthinus* – western yellow bat. NatureServe Infonatura (webpage) http://www.natureserve.org/infonatura. Accessed April 11, 2006.

Nevada Department of Wildlife. 2005. Identification of species of conservation priority. State of Nevada Comprehensive Wildlife Conservation Strategy. p. 59.

O'Farrell, M.J., J.A. Williams, and B. Lund. 2004. Western yellow bat (*Lasiurus xanthinus*) in Southern Nevada. The Southwestern Naturalist 49:514-518.

O'Farrell Biological Consulting. 2006. Final report: Long-term acoustic monitoring of bat populations associated with an extensive riparian restoration program in Las Vegas Wash, Clark County, Nevada (2004-2005). Prepared for Southern Nevada Water Authority.

Sparks, D.W., M.T. Simmons, C.L. Gummer, and J.E. Duchamp. 2003. Disturbance of roosting bats by woodpeckers and raccoons. Northeastern Naturalist 10:105-108.

Western Bat Working Group. 1998. Regional bat species priority matrix. Western Bat Working Group (webpage). http://www.wbwg.org/spp_matrix.html. Accessed April 6, 2006.

Williams, J.A. 2001. Community structure and habitat use by bats in the upper Moapa Valley, Clark County, Nevada. Unpublished M.A.S. Thesis. University of Nevada, Las Vegas. p. 55.

COLORADO RIVER COTTON RAT (*Sigmodon arizonae plenus*)

Introduction

The Colorado River cotton rat (*Sigmodon arizonae plenus*) is a subspecies of the Arizona cotton rat (*Sigmodon arizonae*), although previously it was considered to be a subspecies of the hispid cotton rat (*Sigmodon hispidus*). In 1970, karyotypical and morphological evidence was used to name the Colorado River cotton rat as a subspecies (Sevringhaus and Hoffmeister 1978, Zimmerman 1970). In the literature, it is referred to by all of the above names. Prior to 1970, many references to *S. hispidus*, and specifically *S. hispidus plenus*, the subspecies which used to encompass both this species and the Yuma hispid cotton rat (currently known as *S. hispidus eremicus*), may have been *S. arizonae plenus*.

Legal Status

The Colorado River cotton rat is Federally listed as a Candidate 2 species and as a species of special concern in the state of California (Blood 1998). It is also classified as an endangered species by the IUCN (World Conservation Union) (Nowak 1997, Blood 1998).

Distribution

The Arizona cotton rat has a current distribution that includes southeastern California, Arizona, and western Mexico and historically extended up to extreme southern Nevada (Hoffmeister 1986). The exact current and historical distributional range of the Colorado River cotton rat is not well known. In Nevada, the first records of *Sigmodon* spp. in the state come from a marsh in the extreme southern portion of the state from data collected in 1934 (Hall 1946). An individual *Sigmodon* spp. was captured in 1961 in this same marsh, which subsequently dried up. Further capture efforts in 1966 were unsuccessful and the species has been considered extirpated in the state since this date (Bradley 1966).

Further south, records of the Colorado River cotton rat are more common. Individuals have been documented in Arizona from Parker to Ehrenburg (Hoffmeister 1986). In California, Goldman (1928) reported the species in three locations: 1) Needles, California, 2) near Parker, Arizona, and 3) 15 miles southwest of Ehrenburg, Arizona. The California Department of Fish and Game has recorded the species in Imperial, Riverside, and San Bernardino counties, from Palo Verde to Needles, based on investigations carried out by Blood and Huckaby (Williams 1986). Andersen and Nelson (1998) conducted trapping for a variety of small mammal species at three sites along the Colorado River and at each site several Colorado River cotton rats were captured. The survey sites included a site recently re-vegetated on the Cibola National Wildlife Refuge, No-Name Lake near Parker, Arizona, and at one site on the Bill Williams National Wildlife Refuge. All sites were located on the eastern (Arizona) side of the river. Studies suggest that, within its area of occurrence, the presence of the Colorado River cotton rat is isolated and spotty, rather than continuous.

The southern extent of the range of this species is still unknown. In work conducted by Blood (1990), a qualitative analysis found this species occurring north of the Palo Verde Mountains, along the LCR, and the Yuma Hispid Cotton Rat occurring south of the mountains. It was concluded that a good estimate of species affinity was locality because the two species are allopatric (Blood 1990). There is no conclusive data indicating that the two species do not overlap in any part of their range, and there is evidence indicating that they may be expanding their ranges into agricultural areas (Blood 1998). Zimmerman (1970) hypothesized that, in some areas, this species may become sympatric with the Yuma hispid cotton rat and, given the possible expansion of both species into agricultural areas, this may be now occurring.

Life History

General Description

Little information has been collected on Colorado River cotton rat life history and habitat requirements, although there is an assumption that they are similar to other subspecies of the Arizona cotton rat and the closely related hispid cotton rat (California Department of Fish and Game 2005). It is known to feed primarily on grasses (California Department of Fish and Game 2005). Other species, such as the *S. hispidus*, are known to be much more generalist, eating crops, some insects, eggs, and carrion. *S. arizonae* may vary their diet with some of these items as well (California Department of Fish and Game 2005).

Originally, this species was considered to be associated strongly with marsh vegetation (Goldman 1928), but further research has found the species to inhabit a greater variety of habitats. The species is known to use riparian thickets with moderate to dense grass cover, but may also use drier grassy areas (Hoffmeister 1986). It has also been found in areas associated with common reed (*Phragmites communis*) (Zimmerman 1970).

Identification

Identification of this species is very difficult due to its possibly overlapping distribution with the Yuma hispid cotton rat, and the inability to distinguish the two species using external features. In the absence of chromosomal data, it has been determined that only a combination of morphological characteristics can be used to separate the Colorado River cotton rat from the Yuma hispid cotton rat (Blood 1990). Hind leg length, combined with several skull measurements and features, may be useful in species identification. An Arizona cotton rat was bred in the laboratory with a hispid cotton rat and produced only infertile young (Zimmerman 1970). This would suggest that hybridization is rare, if it occurs at all.

Breeding

No specific information exists on the breeding habits for the Colorado River cotton rat but it is assumed to be similar to other species of *Sigmodon* (California Department of Fish and Game 2005). *Sigmodon* spp. are known to breed throughout the year in the southern portion of their range, and the young of *Sigmodon arizonae* have been collected during almost every month of the year (Nowak 1997, Hoffmeister 1986). Cotton rats are known to construct nests of woven grass either in burrows or on the ground (Baar et al. 1974). They do not migrate and are active both diurnally and nocturnally (California Department of Fish and Game 2005).

Habitat

In capture studies conducted by Andersen and Nelson (1998), the Colorado River cotton rat was most readily captured in grass/cattail communities. In restoration sites, this species was not captured until an herbaceous understory had developed. Andersen and Nelson (1998) concluded that the development of an understory may be the most important element in providing quality habitat for many species of small mammals, including *S. arizonae*. In preliminary presence/absence studies conducted by the Bureau of Reclamation at the Cibola Nature Trail restoration site on the Cibola National Wildlife Refuge in 2004-2005, *Sigmodon* species were only captured in areas with a dense understory of Johnsongrass (*Sorghum halapense*) (Reclamation, unpublished data).

Backwater habitat along the lower Colorado River has been altered by channelization, agricultural use, and storage of water, invasion by saltcedar, and decreased flow regimes due to dam construction. These alterations all may have contributed to a decline in the population of lower Colorado River cotton rats (Bradley 1966, Williams 1986, Andersen and Nelson 1999). There are also some indications that the species, along with the Yuma hispid cotton rat, may be expanding its population and range into agricultural lands (Blood 1998).

Threats

There does not appear to be any immediate threat to the continued survival to the Colorado River cotton rat as a geographically distinct subspecies (Blood 1998). Little information is available describing range and habitat use and further research is needed. Once the distributional range and habitat types used by this species are determined, heretofore unknown threats may be identified.

LCR MSCP Conservation Measures

CRCR1 — Conduct research to better define Colorado River cotton rat habitat requirements. Conduct research, if needed, to better define elements of Colorado River cotton rat habitat and provide information necessary to design and manage created habitat.

CRCR2 — Create 125 acres of Colorado River cotton rat habitat. Of the 512 acres of marsh to be created for Yuma clapper rail, at least 125 acres will be designed to also provide Colorado River cotton rat habitat in reaches 3 and 4 near occupied habitat. Additional habitat may be provided by marsh vegetation that establishes along margins of the 360 acres created backwaters.

LCR MSCP Research and Monitoring Needs

Research priorities for the Colorado River cotton rat include: 1) delineate distributional range, 2) determine habitat use, 3) determine Colorado River cotton rat density within occupied habitats, 4) determine vegetation characteristics of preferred habitats, and 5) identify limiting factors influencing habitat selection (i.e., availability of certain food types, soil moisture, vegetation density). Blood (1998) recommends investigating distribution and population density of *S. a. plenus* in Arizona and California for a minimum of 3 years, and determining the extent of suitable habitat available for the species. Preliminary data, recorded during the winter of 2004-2005, indicate that this species is present in created habitat with a dense ground layer of vegetation (Reclamation, unpublished data). This may indicate that riparian restoration efforts that were not originally thought to provide habitat for this species may actually be providing unexpected habitat benefits for both this species and the Yuma hispid cotton rat. The LCR MSCP conservation measures focus solely on creation of marsh habitat to provide for the habitat needs of this species. It may be possible to create suitable habitat for the Colorado River cotton rat in cottonwood-willow and mesquite habitat creation sites.

Literature Cited

Andersen, D.C. 1994. Demographics of small mammals using anthropogenic desert riparian habitat in Arizona. Journal of Wildlife Management 58(3).

Andersen, D.C., and M. Nelson. 1999. Rodent use of anthropogenic and 'natural' desert riparian habitat, lower Colorado River, Arizona. Regulated Rivers: Research & Management 15:377-393.

Baar, S.L., E.D. Fleharty, and M.F. Artman. 1974. Utilization of deep burrows and nests by cotton rats in west-central Kansas. Southwestern Naturalist 19:440-444

Blood, B.R. 1990. Taxonomy and distribution of *Sigmodon* in California. Bulletin of the Southern California Academy of Sciences 89(2):86-93.

Blood, B.R. 1998. Arizona cotton rat (*Sigmodon arizonae*) Mearns 1890. *In:* D.J. Hafner, E. Yensen, and G.L. Kirkland (editors), North American rodents, status, survey, and conservation action plan. IUCN/SSC Rodent Specialist Group, IUCN, Gland, Switzerland and Cambridge, UK. 171 p.

Bradley, G.W. 1966. Status of the cotton rat in Nevada. Journal of Mammalogy Vol. 47, No. 2.

California Department of Fish and Game. 2005. Arizona cotton rat. http://www.dfg.ca.gov/whdab/html/M124.html. Accessed on October 2005.

Goldman, E.A. 1928. Three new rodents from western Arizona. Proceedings of the Biological Society of Washington 41:203-206.

Hall, R.E. 1946. Mammals of Nevada. University of Nevada Press. Reno, Nevada.

Hoffmeister, D. 1986. Mammals of Arizona. University of Arizona Press. Tucson, Arizona. 564 p.

Nowak, R.M. 1997. Walkers Mammals of the World – Online edition. John Hopkins University Press http://www.press.jhu.edu/books/walkers_mammals_of_the_world/rodents. Accessed October 2005.

Sevringhaus, W.D., and D. Hoffmeister. 1978. Qualitative cranial characters distinguishing *Sigmodon hispidus* and *Sigmodon arizonae* and the distribution of these two species in northern Mexico. Journal of Mammalogy Vol. 59, No. 4.

Williams, D.F. 1986. Mammalian species of special concern in California. Wildlife Management Division Administrative Report 86-1.

Zimmerman, E.G. 1970. Karyology, systematics and chromosomal evolution in the rodent genus *Sigmodon*. Publications of the Museum, Michigan State University Biological Series 4:389-454.

YUMA HISPID COTTON RAT
(*Sigmodon hispidus eremicus*)

Introduction

The Yuma hispid cotton rat (*Sigmodon hispidus eremicus*) is a geographically isolated subspecies of the hispid cotton rat (*S. hispidus*). It is known to occur only in the extreme southern section of the Lower Colorado River (LCR) within the United States and, presumably, in the adjacent areas of Mexico. The Yuma hispid cotton rat is one of two species of *Sigmodon* known to occur along the LCR; the other is the Colorado River cotton rat (*Sigmodon arizonae plenus*), which is a subspecies of the Arizona cotton rat (*S. arizonae*). Both species (and the respective LCR subspecies) are very similar and can only be reliably separated through differences in skull morphology or by DNA analysis. These two species were determined to be separate species in 1970 and many historical references to hispid cotton rats along the LCR prior to 1970 may refer to the Colorado River cotton rat, instead of the Yuma hispid cotton rat.

Legal Status

The Yuma hispid cotton rat is a Federal C2 candidate taxa. It is also in the IUCN Red List category and is considered a lower risk, near-threatened species. In California, it is a taxon of concern (Hafner and Kirkland 1998).

Distribution

The distribution of the Yuma hispid cotton rat is considered to be restricted to areas along the LCR, south of the Palo Verde Mountains, and small, isolated areas of suitable habitat west of Yuma, Arizona, in Imperial County, California (Blood 1990). Although the Yuma hispid cotton rat may have historically occurred in the western part of the Gila River Valley east of Yuma, no evidence exists indicating that the Yuma hispid cotton rat is currently present in these areas (Hoffmeister 1986). The Yuma hispid cotton rat has been trapped in Mexico close to the border, near Yuma, Arizona (Blood 1990). It is presumed that it was once much more prevalent in the Colorado River Delta area before changes on the river brought about the end of river flows reaching the delta (Hafner and Kirkland 1998).

The hispid cotton rat (*S. hispidus)* is very widespread and its range includes northern South America, Mexico, Central America, and the southeastern and south central United States. However, the Yuma (*S. h. eremicus*) subspecies is geographically isolated from the rest of the species, with the closest population of hispid cotton rats located in southeastern Arizona (Cameron and Spencer 1981).

The range of the subspecies is limited to areas near Yuma, Arizona, but may overlap with that of the Colorado River cotton rat, which also occurs along the LCR; the southern extent of its range is unknown. The northern extent of the range of the Yuma hispid cotton rat is presumed to be the Palo Verde Mountains, but there is not enough data to assume that no overlap occurs between the two species (Blood 1990). There is evidence that both species may be expanding their ranges into agricultural areas, and this range expansion may result in some overlap (Blood 1998).

Hoffmeister (1986) observed both species (*S. hispidus* and *S. arizonae)* together in laboratory situations and observed the hispid cotton rat to be subordinate. In the laboratory, the two species were aggressive, with the Arizona cotton rat taking a dominant position over the hispid cotton rat by gaining first access to food and other resources. Hoffmeister hypothesized that this subordination may be a limiting factor in the range distribution of the hispid cotton rat in Arizona.

Life History

General Description

There is not a great deal of information available specific to the life history of the Yuma hispid cotton rat. The Yuma subspecies is not considered to be markedly differentiated from the main species, which occurs in eastern Arizona and likely shares most of the life history traits with the rest of the species (Hoffmeister 1986).

The hispid cotton rat has a small home range, with females typically occupying a home range of 0.1 to 0.3 ha. and males occupying a home range of 0.4 to 0.5 ha (Hawthorne 1994). Radio-telemetry studies have shown that hispid cotton rats utilize these home ranges in a systematic manner, over multiple days. This may allow the species to utilize areas with patchy distribution of needed resources in a more efficient manner (Cameron 1995).

Hispid cotton rats are active all year, feeding mainly on grasses, and insects only on a seasonal basis. Grass height and density have been documented as important habitat components for hispid cotton rats (Cameron and Spencer 1981); they utilize runways through dense herbaceous growth and nests are built of woven grass. Population density is regulated by avian predators; mammal predation is considered to be incidental. Some of the principal competitors for resources for hispid cotton rats include other cricetid rodents, especially microtines, and the common mouse (*Mus musculus*) (California Department of Fish and Game 2005).

Identification

Identification of this species is difficult due to its distribution possibly overlapping that of the Colorado River cotton rat range, and the inability to distinguish the Yuma hispid cotton rat from the Colorado River cotton rat using external features. In the absence of chromosomal data, it has been determined that only a combination of morphological characteristics (hind leg length and skull measurements) can be used to separate the

Yuma hispid cotton rat from the Colorado River cotton rat (Blood 1990, Sevringhaus and Hoffmeister 1978, Zimmerman 1970). An Arizona cotton rat was bred in the laboratory with a hispid cotton rat and produced only infertile young (Zimmerman 1970). This would suggest that hybridization is rare, if it occurs at all.

Peppers and Bradley (2000) conducted genetic analysis on eight subspecies of *Sigmodon* and determined that *S .h. eremicus* is genetically similar to other subspecies of *Sigmodon*, which occur in New Mexico and Oklahoma. The Yuma hispid cotton rat was placed in a distinct clade with these two other species but the trichotomy of these three subspecies within the clade remained unresolved. More work may be needed to determine the exact genetic differentiation between the *eremicus* subspecies and the other closest *S. hispidus* subspecies.

Breeding

Hispid cotton rats breed throughout the year. In Louisiana, they have been observed to have a gestation period of 27 days and produce precocial young, which are weaned after 15 to 25 days. The average litter size is 5.6 young, and one captive female in a laboratory situation was recorded to have produced nine litters in a 10-month period (Hoffmeister 1986). Breeding starts after 2-3 months of age, and the average life span is 6 months (Cameron and Spencer 1981).

S. hispidus have exhibited bimodal population fluctuations throughout the year in other parts of its southern North American range (Texas, Georgia, Mexico). Maximum densities have been recorded in the fall, with smaller population peaks occurring in spring. The lowest densities have been documented to occur in winter and summer (Cameron and Spencer 1981).

LCR MSCP Conservation Measures

YHCR1 — Conduct research to better define Yuma hispid cotton rat habitat requirements. Conduct research, if needed, to better define the elements of Yuma hispid cotton rat habitat and provide information necessary to design and manage created habitat.

YHCR2 — Create 76 acres of Yuma hispid cotton rat habitat. Of the 5,940 acres of cottonwood-willow to be created as habitat for covered species, at least 76 acres will be designed to provide habitat for the Yuma hispid cotton rat in reaches 6 and 7 near occupied habitat. Created Yuma hispid cotton rat habitat will be designed and managed to support a moist herbaceous understory, an element of the species' habitat.

LCR MSCP Research and Monitoring Needs

Hafner and Kirkland (1998), in their description and profile of the hispid cotton rat, list two recommended actions that apply specifically to *S. h. eremicus*. These recommendations are: 1) conduct surveys to determine population status and distribution of *S. h. eremicus*, with particular focus on habitat loss in the Colorado River Delta and range expansion into agricultural areas of the lower Colorado River Valley, and 2) to evaluate the systematic relationships of isolated subspecies of *S. hispidus* to neighboring conspecifics using biochemical and genetic analysis.

Important research priorities have been established, including: 1) what habitats are being utilized by this species, 2) in what density they are found in utilized habitats, 3) how density of Yuma hispid cotton rats compares between restored habitats and other areas (agricultural areas and natural occurring sites of native vegetation), 4) the distributional range of the species, and if the range overlaps with that of the Colorado River cotton rat, and 5) the actual genetic differentiation between the Yuma subspecies of cotton rat and the nearest population of the cotton rat.

There is a good deal of evidence that this species occurs in thick grassy habitats that occur naturally in many restored cottonwood-willow habitats. Through the efforts to restore cottonwood-willow habitats as part of the LCR MSCP program, there may be a great deal of suitable habitat created for the Yuma hispid cotton rat. A variety of habitats need to be sampled for presence/absence and density within commonly occupied habitat types. Various areas along the LCR need to be sampled to determine what habitat types are used by Yuma hispid cotton rats. This can be accomplished with presence/absence live trapping. Density arrays can then be established in areas known to have Yuma hispid cotton rats to determine how restored sites compare to other sites used by the species. This information could then be used to determine range boundaries for the species.

Finally, it would be helpful to understand the exact genetic relationship between *S. hispidus eremicus* and other *S. hispidus* populations, specifically those populations which occur in southeastern Arizona. Hoffmeister (1986) hypothesized that there may be little in the way of genetic differentiation between the *eremicus* subspecies and the main species group. An understanding of the exact genetic differentiation of the subspecies would help in determining how close the subspecies is to the main species. Because a great deal of information exists on habitat use and diet of *S. hispidus* in other locations, information on the genetic differentiation could be used to determine if this subspecies can be considered similar to the main species, or a highly differentiated subspecies with possibly unique life history and habitat use characteristics. This information could then be used, in combination with data collected from trapping, to determine what restoration efforts may meet the needs of this species. It would also allow us to determine the priority of this subspecies within the entire LCR MSCP covered species list.

Other Potential Research and Monitoring Opportunities

Hafner and Kirkland (1998) recommend conducting surveys to determine population status and distribution within the Colorado River Delta in Mexico. Habitat alteration within the Delta may affect population trend.

Literature Cited

Andersen, D.C. 1994. Demographics of small mammals using anthropogenic desert riparian habitat in Arizona. Journal of Wildlife Management 58(3):445-454

Andersen, D.C., and M. Nelson. 1999. Rodent use of anthropogenic and 'natural' desert riparian habitat, lower Colorado River, Arizona. Regulated Rivers: Research & Management 15:377-393.

Blood, B.R. 1990. Taxonomy and distribution of *Sigmodon* in California. Bulletin of the Southern California Academy of Sciences 89(2):86-93.

Blood, B.R. 1998. Arizona cotton rat (*Sigmodon arizonae*) Mearns 1890. *In:* D.J. Hafner, E. Yensen, and G.L. Kirkland (editors), North American rodents, status, survey, and conservation action plan. IUCN/SSC Rodent Specialist Group, IUCN, Gland, Switzerland and Cambridge, UK. 171 p.

Cameron, G.N. 1995. Temporal use of home range by the hispid cotton rat. Journal of Mammalogy 76(3):819-827.

Cameron, G.N., and S.R. Spencer. 1981. *Sigmodon hispidus*. Mammalian species. No. 158, p.1-9. (website) http://www.science.smith.edu/departments/Biology/VHAYSSEN/msi/.

Hafner, D.J., and G.L. Kirkland, Jr. 1998. Hispid cotton rat (*Sigmodon hispidus*) Say and Ord 1825. *In:* D.J. Hafner, E. Yensen, and G.L. Kirkland (editors), North American rodents, status, survey, and conservation action plan. IUCN/SSC Rodent Specialist Group, IUCN, Gland, Switzerland and Cambridge, UK. 171 p.

Hawthorne, D.W. 1994. Cotton Rat. *In:* S. Hygnstrom, R. Timm, and G. Larson (editors), Prevention and Control of Wildlife Damage-1994 USDA-APHIS, Animal Damage Control, Cooperative Extension Division, Institute of Agriculture and Natural Resources, University of Nebraska, Lincoln.

Hoffmeister, D. 1986. Mammals of Arizona. University of Arizona Press. Tucson, Arizona. 564 p.

Peppers, L.L., and R.D. Bradley. 2000. Cryptic species in *Sigmodon hispidus*: Evidence from DNA sequences. Journal of Mammalogy 81(2):332-243.

Sevringhaus, W.D., and D. Hoffmeister. 1978. Qualitative cranial characters distinguishing *Sigmodon hispidus* and *Sigmodon arizonae* and the distribution of these two species in northern Mexico. Journal of Mammalogy Vol 59, No. 4.

Williams, D.F. 1986. Mammalian species of special concern in California. Wildlife Management Division Administrative Report 86-1.

Zimmerman, E.G. 1970. Karyology, systematics and chromosomal evolution in the rodent genus *Sigmodon*. Publications of the Museum, Michigan State University Biological Series 4:389-454.

LEAST BITTERN
(*Ixobrychus exilis*)

Introduction

The least bittern (*Ixobrychus exilis)* inhabits freshwater and brackish marshes and breeds in low-lying areas associated with large rivers, lakes, and estuaries. It is found from southeastern Canada to South America (Gibbs et al. 1992, American Ornithological Union 1998). *Ixobrychus exilis* is one of five superficially similar subspecies distributed throughout the least bittern's range (Hancock and Kushlan 1984). The least bittern was previously classified into eastern (*exilis*) and western (*hesperis*) subspecies (Palmer 1962), but recent data on plumage and morphology do not support this dichotomy (Dickerman 1973).

Legal Status

The least bittern is listed as a species of special concern in Arizona and California (Remsen 1978, Arizona Game and Fish 2001). It is listed by the U.S. Fish and Wildlife Service as a migratory nongame bird of management concern (USFWS 1995). It is on the Audubon Society's Blue List, which is published in their ornithological field journal *American Birds* (Tate 1986). It is not listed by Nevada or Mexico.

Distribution

Historical Range

Bent (1926) listed the breeding range of the least bittern from New Brunswick to southern Ontario, through North Dakota and Minnesota, south to Central Mexico, Puerto Rico, and Jamaica, Tule Lake in Oregon and California, south to Baja California, western Mexico, and southwestern Guatemala. The wintering range in the United States includes Florida (Bent 1926). It also extends south from Fort Verde in Arizona to the islands of the Caribbean, as well as Central and South America to Patagonia (Bent 1926).

As of 1914, the only record for the least bittern in Arizona was one occurrence on the Colorado River, between forts Yuma and Mohave, in September 1865 (Swarth 1914). A least bittern was captured in a muskrat trap near Topock, Arizona, in December 1943, indicating possible wintering along the lower Colorado River (Monson 1949). Monson and Phillips (1981) listed the least bittern as a fairly common resident in the Colorado River Valley, north to Topock, and as a resident in the cattails along the Salt River in the Phoenix area. In California, Grinnell (1915) listed it as a fairly common summer visitant north through the interior of the Sacramento Valley and also occurring along the southern coast. In 1940 and 1941, they were encountered near Bard, California, in the fall and winter months and in April and May near Yuma and Somerton, Arizona (Arnold 1942). In Nevada, Linsdale (1951) listed the least bittern as infrequent in summer and reports it from Washoe, Elko, Churchill, Esmeralda, and Clark counties. The first records for southern Nevada were from Lake Mead, September 1938 (Grater 1939). It was recorded from the Great Salt Lake in the 1880s (Ryser 1985). The first record for southern Utah was in May 1938, along the Virgin River at Saint George (Hardy 1939). The next sightings were in June and August 1965, along the Virgin River, south of Washington (Russell 1967).

Current Range

The breeding range of the least bittern is now from southeastern Canada, through the United States and Mexico to Costa Rica (American Ornithological Union 1998). It is discontinuous between the Mississippi River Valley and the Pacific states (Gibbs et al. 1992). Western populations are concentrated in low-lying areas of the Central Valley and Modoc Plateau in California, the Klamath and Malheur basins of Oregon, along the Colorado River in southwestern Arizona and southeastern California, the Salton Sea area, and the Ciénega de Santa Clara in Mexico (Rosenberg et al. 1991, Gibbs et al. 1992, Patten et al. 2003, Hinojosa-Huerta et al. 2004). Breeding has recently been confirmed in central, south central, and southeastern Arizona, as well as southern Nevada (Corman 2005, Branca 2005).

The winter range is primarily south of areas with prolonged winter frosts: along the Atlantic coastal plain from Maryland and Virginia, south to Louisiana and Texas, with peak numbers in southern Florida, the Rio Grande Valley, the lower Colorado River Valley, and Baja California (Palmer 1962, Hancock and Kushlan 1984, Root 1988, Gibbs et al. 1992). Many also overwinter in the Greater Antilles, east and Central America, and south to Panama (Gibbs et al. 1992).

Populations within LCR MSCP Project Boundary

Rosenberg et al. (1991) stated that the largest populations along the lower Colorado River are in extensive cattail (*Typhus* spp.) and bulrush (*Scirpus* spp.) marshes, such as at Topock and near Imperial Dam. The Arizona Breeding Bird Atlas states that, in much of southwestern Arizona at elevations below 1000 ft (305 m), extensive cattail marshes often harbor least bitterns (Corman 2005). In 2006, presence/absence surveys for Yuma clapper rail (*Rallus longirostris yumanensis*) along the lower Colorado River were modified to include surveys for least bittern, California black rail (*Laterallus jamaicensis coturniculus*), and Virginia rail (*R. limicola*) (USFWS 2006). Population information from these surveys is not immediately available.

In reaches 1 and 2, the least bittern is listed as a rare transient in the Lake Mead National Recreation Area (Blake and USNPS 1978). One was heard in June 2005, near the inflow of the Muddy River into Lake Mead (J. Barnes per. comm.).

The bird checklists for Havasu, Bill Williams River, Cibola, and Imperial National Wildlife Refuges all list the least bittern as common, certain to be seen in suitable habitat, from June through August (USFWS 1994, 1995). In Reach 3, the majority of marsh habitat is located in Topock Marsh, Topock Gorge, and the Bill Williams River Delta. In reaches 4 and 5, there is scattered marsh habitat through the Colorado River Indian Tribes (CRIT) Reservation, Cibola and Imperial NWRs, and near Imperial Dam. In Reach 6, the only extensive marsh habitat is at Mittry Lake. Reach 7 lacks habitat for the least bittern.

Life History

General Description

The least bittern is the smallest member of the heron family and is one of the most inconspicuous of all marsh birds (Weller 1961, Gibbs et al. 1992). It's very small size (28-36 cm length, 43 cm wing span, 80 g weight) and contrasting color pattern are diagnostic field marks. Sexes are similar in size but the plumage is dimorphic. The crown, back, and tail on the male are greenish black, while those of the female are a purple-chestnut. The neck, sides of the body, and underparts are brown and white, with the neck of the female darkly streaked. The wings are chestnut with contrasting and conspicuous pale patches and the wingtips are slate. The bill is thin and yellow; legs and feet are a straw to buffy yellow. Plumage of the juvenile is similar to the female with the crown and back a lighter brown; the chest and throat have a striped appearance (Palmer 1962, Gibbs et al. 1992, Sibley 2000). Newly hatched chicks are covered with a pale buff down, new feathers start to emerge at 8 days, and juvenile plumage is nearly complete at 36 days (McVaugh 1975).

Males vocalize a dovelike coo that is repeated 3-5 times, often in spring, and is thought to advertise their presence (Gibbs et al. 1992, Monfils 2003). A gack-gack call is also heard and is given from the nest, perhaps as a contact call between mates (Weller 1961). These two calls are the ones most often heard during marsh bird surveys.

Breeding

In Arizona, resident least bitterns likely begin breeding before migratory populations, with males initiating their cooing calls in March and April (Corman 2005). Rosenberg et al. (1991) describes them as a locally common breeder from April through September. Throughout their entire range, nests are typically built among dense stands of emergent or woody vegetation (typically *Typha*, *Carex*, and *Scripus*, occasionally *Phragmites, Sagittaria, Salix*, *Cephalanthus,* and *Rhizophora*) (Weller 1961, Palmer 1962). The nest platform and canopy is constructed primarily by the male and is made by pulling down and crimping surrounding vegetation; short stems and sticks are added in a spoke-like manner to form the nest (Weller 1961). Nests are well concealed and are 15-20 cm in diameter and 5-12 cm in depth (Nero 1950). They are usually 15-76 cm above water that is 8-96 cm in depth (Weller 1961, Gibbs et al. 1992). Nests are usually located adjacent to open water (Weller 1961). Distance from open water can range from 60 cm to 65 m but is usually less than 10 m (McVaugh 1975, Aniskowicz 1981, Gibbs et al. 1992).

Nests have been found scattered throughout suitable habitat or concentrated in loose groupings where the distance between them ranged from 2 to 85 m (McVaugh 1975, Ziebell 1990). Nests have been recorded 1 m apart in highly productive habitat where the least bittern may be considered semi-colonial (Kushlan 1973). Least bitterns have been known to nest within colonies of boat-tailed grackles (*Quiscalus major*) in South Carolina and possibly, in great-tailed grackle colonies in southern Nevada (Post and Seals 1993, J. Healy pers. comm.)

Typical clutches are 4-5 eggs (range 2-7) and the time from laying the first egg to the hatching of the first egg ranges from 19 to 21 days (Bent 1926, Weller 1961, Bogner and Baldassarre 2002). Both sexes incubate the eggs, the female perhaps more than the male (Weller 1961). The chicks are born semialtricial (downy and need to be fed by adults) and nidicolous (they stay at the nest) (Weller 1961, Erlich et al. 1988). The young are fed by regurgitation and by the males more than the females (Weller 1961). They can forage on their own within 1-2 weeks (Nero 1950). The young normally leave the nest permanently by 13-15 days but linger nearby for 1-2 weeks (Nero 1950, Palmer 1962). Approximate age at first flight is 29 days (Bogner and Baldassarre 2002). The least bittern will renest and double brood (Post and Seals 1998, Bogner and Baldassarre 2002).

Diet

The least bittern's major food items are small fish and insects (Gibbs et al. 1992). Stomachs of 20 least bitterns collected at Imperial NWR contained (in order of importance): freshwater shrimp (*Paleomonetes* spp.), crayfish (*Procambarus clarkia*), bluegill (*Lepomis macrochirus*), mosquitofish (*Gambusia affinis*), and threadfin shad (*Dorosoma petenense*) (Martinez 1994). Frogs (*Rana* spp.) are also a prey item (Weller 1961). Insects taken are mainly Odonata and Orthoptera (Gibbs et al. 1992). Least bitterns may also prey on the eggs and young of yellow-headed blackbirds (*Xanthocephalus xanthocephalus*; Roberts 1936 *in* Gibbs et al., J. Healy per. comm.).

The least bittern's small size, highly compressed trunk, and ability to grasp with its feet enable it to move through dense vegetation. It forages by clinging to emergent vegetation over open water and extending its long neck, wading along the edge of open water, and using small constructed foraging platforms at rich feeding sites (Eastwood 1932, Sutton 1936, Weller 1961, Gibbs et al. 1992). Foraging behaviors used by the least bittern are standing in one place, walking slowly, moving its head back and forth, and flicking its wings to startle prey (Kushlan 1978).

Habitat

In general, the least bittern occupies freshwater and brackish marshes with dense, tall growths of emergent vegetation, interspersed with clumps of woody vegetation and open water (Gibbs et al. 1992). Surveyors for the Arizona Breeding Bird Atlas found least bitterns in marshes along rivers, ponds, lake edges, and less frequently, along irrigation and runoff ditches from agricultural areas (Corman 2005). In the lower Colorado River Valley, generally marshes that are dominated by dense cattails or bulrushes support large numbers of breeding insectivorous wading birds, including rails and least bitterns (Rosenberg et al. 1991). Least bitterns at the Salton Sea reach peak abundance along rivers and wide irrigation ditches, particularly in dense stands of southern (*Typha domingensis*) and broad-leaved cattails (*T. latifolia*), but some may use common reed

(*Phragmites australis*) or saltcedar (*Tamarix ramosissima*) if cattails are nearby (Patten et al. 2003).

Nests have been recorded in a variety of vegetation types and over varying depths of water (Table 1). Some sawgrass (*Cladium jamaicense*) and cattail habitat were 2 m in height (Kushlan 1973, McVaugh 1975).

Table 1. Depth of water and vegetation type at least bittern nest sites

Depth of water	**Vegetation type**	**Author(s)**
40 cm	Cattail	Aniskowicz 1981
60 cm	Cattail	Nero 1951
12-40 cm	Cattail	Post and Seals 1993
8-97 cm	Cattail and bulrush	Weller 1961
30 cm	Cattail	McVaugh 1975
5, 29 cm	Cattail	Manci and Rusch 1988
50 cm	Dense vegetation	Fredrickson and Reid 1986 in Gibbs et al. 1992

The least bittern is most regularly found in wetlands greater than 5 ha in Iowa,, suggesting the species may be area-sensitive, but territorial individuals in Maine have been seen on wetlands as small as 0.40 ha (Brown and Dinsmore 1986, Gibbs and Melvin 1990 *in* Gibbs et al. 1992). The density of least bitterns that nested within boat-tailed grackle colonies in South Carolina were 3 pairs/ha in a 13-ha study area and 12 pairs/ha in a 3.3-ha study area (Post and Seals 1993). Weller (1961) located 62 nests in a 33.5-ha study area but some of these might have represented renests or second nests. The breeding density of the least bittern in some marshes along the lower Colorado River has been estimated at 40 birds/40 ha (Rosenberg et al. 1991). In Iowa, they were most abundant in freshwater marshes when ratios of emergent vegetation cover were equal to open water, also known as the hemi-marsh condition (Weller and Spatcher 1965 *in* Gibbs et al. 1992). Gibbs and Melvin (1992a) stated that the preservation of wetlands greater than 5 ha with dense, tall (>1 m) emergent vegetation over relatively deep water (10-50 cm) and interspersed with patches of open water was the most important management need for the least bittern.

Threats

Least bittern often nest over water and away from shore, making them less vulnerable to land predators (Bogner and Baldassarre 2002). Snapping turtles (*Chelydra serpentine*) and red-tailed hawks (*Buteo jamaicensis)* have been identified as predators of adult least bitterns within its range (Trautman 1940 *in* Gibbs et al. 1992, Weller 1961). Snakes, turtles, crows, raptors, and raccoons (*Procyon lotor*) have been documented as predators of chicks and eggs (Bent 1926). The marsh wren (*Cistothorus palustris*) has been suspected of puncturing eggs (Ziebell 1990, Bogner and Baldassarre 2002).

Nesting aquatic birds that feed on fish and invertebrates along the lower Colorado River may be bioaccumulating potentially toxic concentrations of selenium in their tissues and eggs (King et al. 2000). Least bitterns collected at the Imperial NWR had selenium concentrations in their livers above the 3-ppm threshold for which toxic effects on predatory fish or birds might be expected (Martinez 1994).

Destruction of wetland habitat is likely the greatest threat to the least bittern nationwide (Gibbs et al. 1992). Changes in points of diversion in reaches 3-5 would lower groundwater levels sufficiently in those reaches to reduce the extent or quality of 53.7 ha (133 ac) of habitat provided by marshes associated with backwaters. Up to 28.3 ha (70 ac) of habitat could be removed to maintain channel functions (e.g., dredging desilting basins) in the river and 12.1 ha (30 ac) in irrigation ditches (LCR MSCP 2004a). The least bittern prefers tall, dense growths of emergent vegetation (Gibbs et al. 1992). Burning decadent emergent vegetation to benefit other covered species, such as the Yuma clapper rail, will result in a temporary loss of habitat for the least bittern, as will wildfire episodes (LCR MSCP 2004a, Conway and Nadeau 2005).

LCR MSCP Conservation Measures

LEBI1 — Create 512 acres of least bittern habitat.
Create and manage 512 acres (209 ha) of marsh to provide least bittern habitat. This created habitat will also be habitat for the Yuma clapper rail (*Rallus longirostris yumanensis*) (conservation measure CLRA1). Habitat will be created in patches as large as possible. Smaller patches are likely within the range of habitat patch sizes used by the species for foraging and dispersal, and larger patches may be used for breeding. Least bittern habitat will be created and maintained as described in Section 5.4.3.3. Marshes created to provide least bittern habitat will be designed and managed to provide an integrated mosaic of wetland vegetation types, water depths, and open water areas. Priority will be given, when consistent with achieving LCR MSCP goals for other covered species, to establishing habitat near occupied habitat. The largest numbers of least bittern in the LCR MSCP planning area are located at Topock Marsh and the marshes near Imperial Dam, but they are present in suitable marshes throughout the LCR MSCP planning area. Within the mosaic of marsh conditions, least bittern habitat will generally be provided by patches of bulrush and cattails interspersed with small patches of open water that maintain water depths no greater than 12 inches (30.5 cm) (LCR MSCP 2004b).

LCR MSCP Research and Monitoring Needs

Research on the least bittern’s nesting biology, movements, population dynamics, and overwintering biology would help to clarify the status of the species and facilitate its conservation (Gibbs et al. 1992). Along the LCR, research and monitoring priorities include determining distribution, habitat requirements, seasonal movements, habitat maintenance activities, and potential limiting factors affecting population status.

Least bittern surveys have been conducted along the LCR since 2006. Surveys will continue to determine least bittern distribution within the LCR MSCP boundaries. Information gathered can be used to determine the locations of populations in relation to areas where marsh habitat will be created.

Low stem densities and little residual vegetation were considered year-round requisites of Yuma clapper rail habitat in a study in southwestern Arizona (Conway et al. 1993). In Arizona, the least bittern utilizes dense, tall growths of emergent vegetation, interspersed with open water (Corman 2005). Because habitat creation goals assume the use of created habitats by both species, habitat creation projects must be designed to incorporate a mosaic of different habitat characteristics.

Knowledge is incomplete concerning possible migration or seasonal habitat use by least bitterns that breed along the lower Colorado River (Corman 2005). Seasonal movements and habitat use need to be determined so that habitat creation projects will provide habitat requirements needed during non-breeding seasons and to minimize disturbance of potential winter habitats.

Potential habitat management activities for Yuma clapper rails may adversely affect least bitterns in the short term. Controlled burning of marsh habitat has been studied as a potential management tool to reduce large amounts of decadent vegetation and return marshes to early successional growth that is thought to be beneficial to rails (Conway and Nadeau 2005). Least bitterns may be excluded from these marshes until the emergent vegetation reaches a certain density.

Aquatic birds nesting along the lower Colorado River that feed on fish and invertebrates may be bioaccumulating potentially toxic concentrations of selenium in their tissues and eggs (King et al. 2000). It is unknown whether selenium has a detrimental effect on least bittern survival and reproduction.

Literature Cited

American Ornithologist's Union. 1998. Check-list of North American birds, 7th ed. American Ornithological Union, Washington D.C. 829 p.

Aniskowicz, B.T. 1981. Behavior of a male least bittern incubation after loss of mate. Wilson Bulletin 93:395-397.

Arnold, L.W. 1942. Water birds influenced by irrigation projects in the lower Colorado River Valley. Condor 44:183-182.

Arizona Game and Fish Department. 2001. Least bittern (*Ixobrychus exilis*). Unpublished abstract compiled and edited by the Heritage Management System, Arizona Game and Fish Department, Phoenix, Arizona. 8 p.

Bent, A.C. 1926, Life histories of North American marsh birds. U.S. National Museum Bulletin No. 135. 392 p.

Blake, J.G., and U.S. National Park Service. 1978. Birds of the Lake Mead National Recreation Area. Cooperative Nat. Park Ser. Resources Studies Unit, Univ. of Nevada, Las Vegas Dept. of Biological Sciences. Las Vegas, Nevada. 218 p.

Bogner, H.E., and G.A. Baldassarre. 2002. Home range, movement, and nesting of least bittern in western New York. Wilson Bulletin 114:297-308.

Branca, J. 2005. Henderson bird viewing preserve notes. http://list.audubon.org/archives/nvbirds.html. Accessed on August 20, 2005.

Brown, M., and J.J. Dinsmore. 1986. Implications of marsh size and isolation for marsh bird management. Journal of Wildlife Management 50:392-397.

Conway, C.J., W.R. Eddlemen, S.H. Anderson, and L.R. Hanebury. 1993. Seasonal changes in Yuma clapper rail vocalization rate and habitat use. Journal of Wildlife Management 56:282-290.

Conway, C.J., and C.P. Nadeau. 2005. Effects of fire on Yuma clapper rails and California black rails, 2004 Annual Report. Wildlife Research Rpt. No. 2005-01. Arizona Coop. Fish and Wildlife Research Unit, Tucson, Arizona.

Corman, T.E. 2005. Least Bittern (*Ixobrychus exilis*). *In:* T.E. Corman and C. Wise-Gervais (editors), Arizona Breeding Bird Atlas. Univ. of New Mexico Press, Albuquerque. 636 p.

Dickerman, R.W. 1973. The least bittern in Mexico and Central America. Auk 90:689-691.

Eastwood, S.K. 1932. Notes on the feeding of the least bittern. Wilson Bulletin 44:42.

Ehrlich, P.R., D.S. Dobkin, and D. Wheye. 1988. The Birder's Handbook. Simon & Schuster, New York City. 785 p.

Gibbs, J.P., F.A. Reid, and S.M. Melvin. 1992. Least Bittern. *In:* A. Poole, P. Stettenheim, and F. Gill (editors), The Birds of North America, No.17. Philadelphia: The Academy of Natural Sciences, Washington, DC: The American Ornithologist's Union.

Gibbs, J.P., and S.M. Melvin. 1992a. Least Bittern *In:* K. Schneider and D. Pence, (editors), Migratory nongame birds of management concern in the northeastern United States. U.S. Fish & Wildlife Service, Newton Corner, MA.

__________. 1993. Call-response surveys for monitoring breeding waterbirds. Journal of Wildlife Management 57:27-34.

Grater, R.K. 1939. New bird records for Clark County, Nevada. Condor 41:220-221.

Grinnel, J. 1915. A distributional list of the birds of California. Cooper Ornithological Club, Pacific Coast Avifauna, No.11. The Club, Hollywood. 217 p.

Hancock, J., and J. Kuslan. 1984. The heron's handbook. Croom Helm, Ltd., Kent, England.

Hardy, R. 1939. Two new bird records for Utah. Condor 41:86.

Hinojosa-Huerta, O., H. Iturribarría-Rojas, Y. Carrillo-Guerrero, M. de la Garza-Treviño, and E. Zamora-Hernández. 2004. Bird conservation plan for the Colorado River Delta. Pronatura Noroeste, Dirección Sonora. San Luis Río Colorado, Sonora, México.

King, K.A., A.L. Velasco, J. Garcia-Hernandez, B.J. Zaun, J. Record, and J. Wesley 2000. Contaminants in Potential Prey of the Yuma Clapper Rail: Arizona and California, USA, and Sonora and Baja, Mexico, 1998-1999. U.S. Fish & Wildlife Service, Arizona. Ecological Service Field Office, Phoenix, Arizona. 22 p.

Kushlan, J.A. 1973. Least bittern nesting colonially. Auk 90:685-686.

__________. 1978. Feeding ecology of wading birds. National Audubon Society Research Report 7:249-298.

Linsdale, J.M. 1951. A list of the birds of Nevada. Condor 53:228-249.

Lower Colorado River Multi-Species Conservation Program. 2004a. Lower Colorado River Multi-Species Conservation Program, Volume III: Biological Assessment. Final. December 17. (J&S 00450.00) Sacramento, California.

__________. 2004b. Lower Colorado River Multi-Species Conservation Program, Volume II: Habitat Conservation Plan. Final. December 17. (J&S 00450.00) Sacramento, California.

Manci, K.M., and D.M. Rusch. 1988. Indices to distribution and abundance of some inconspicuous waterbirds on Horicon Marsh. Journal of Field Ornithology 59:67-75.

Martinez, C.T. 1994. Selenium levels in selected species of aquatic birds on Imperial National Wildlife Refuge. M.S. Thesis, University of Arizona, Tucson.

McVaugh, W., Jr. 1975. The development of four North American herons (II). Living Bird 14:163-183.

Monfils, M.J. 2003. Special animal abstract for *Ixobrychus exilis* (least bittern). Michigan Natural Features Inventory, Lansing. 6 p.

Monson, G. 1949. Recent notes from the lower Colorado River Valley of Arizona and California. Condor 51:262-265.

Monson, G., and A.R. Phillips. 1981. Annotated checklist of the birds of Arizona, second edition. University of Arizona Press, Tucson. 240 p.

Nero, R.W. 1950. Notes on a least bittern nest and young. Passenger Pigeon 12:3-8.

Palmer, R.S. 1962. Handbook of North American Birds, Volume 1. Yale University Press, New Haven. 567 p.

Patten, M.A., G. McCaskie, and P. Unitt. 2003. Birds of the Salton Sea: Status, Biogeography, and Ecology. University of California Press, Berkeley, Los Angeles, London. 363 p.

Post, W., and C.A. Seals. 1993. Nesting associations of least bitterns and boat-tailed grackles. Condor 95:139-144.

Remsen, J.V., Jr. 1978. Bird species of special concern in California. California Dept. of Fish & Game, Sacramento. 54 p.

Root, T. 1988. Atlas of wintering North American birds, an analysis of Christmas Bird Count data. University of Chicago Press, Chicago.

Rosenberg, K.V., R.D. Ohmart, W.C. Hunter, and B.W. Anderson. 1991. Birds of the lower Colorado River Valley. University of Arizona Press, Tucson. 416 p.

Russell, R.C. 1967. New and additional records of birds in the Virgin River Valley. Condor 69:420-423.

Ryser, F.A., Jr. 1985. Birds of the Great Basin. University of Nevada Press, Reno. 604 p.

Sibley, D.A. 2000. National Audubon Society, The Sibley Guide to Birds. Knopf, New York. 544 p.

Sutton, G.M. 1936. Food capturing tactics of the least bittern. Auk 53:74-75.

Swarth, H.S. 1914. A distributional list of the birds of Arizona. Cooper Ornithological Club, Pacific Coast Avifauna, No. 10. The Club, Hollywood. 133 p.

Tate, J.R. 1986. The blue list for 1986. American Birds 40:227-236.

U.S. Fish and Wildlife Service. 1995. Migratory nongame birds of management concern in the United States: The 1995 list. Office of Migratory Bird Management, Washington, DC. 25 p.

___________. No date. Birds of Havasu National Wildlife Refuge. U.S. Fish and Wildlife Service. Unpaginated. Jamestown, ND: Northern Prairie Wildlife Research Center Online. http://www.npwrc.usgs.govhavaus.htm (Version 22MAY98).

___________. No date. Birds of Bill Williams River National Wildlife Refuge. U.S. Fish and Wildlife Service. Unpaginated. Jamestown, ND: Northern Prairie Wildlife Research Center Online. http://www.npwrc.usgs.govbillwill.htm (Version 22MAY98).

___________. 1994. Birds of Imperial National Wildlife Refuge. U.S. Fish and Wildlife Service. Unpaginated. Jamestown, ND: Northern Prairie Wildlife Research Center Online http://www.npwrc.usgs.govimperial.htm (Version 22MAY98).

___________. 1995. Birds of Cibola National Wildlife Refuge. U.S. Fish and Wildlife Service. Unpaginated. Jamestown, ND: Northern Prairie Wildlife Research Center Online. http://www.npwrc.usgs.govcibola.htm (Version 22MAY98).

___________. 2006. New official survey protocol for Yuma clapper rail surveys. U.S. Fish and Wildlife Service, Arizona Ecological Service Field Office, Phoenix, Arizona.

Weller, M.W. 1961. Breeding biology of the least bittern. Wilson Bulletin 73:11-35.

Ziebell, T.J. 1990. Nesting least bitterns on Rush Lake, Wisconsin. Passenger Pigeon 52:19-28.

CALIFORNIA BLACK RAIL
(*Laterallus jamaicensis coturniculus*)

Introduction

The California black rail (*Laterallus jamaicensis coturniculus*) is one of five subspecies of the black rail that are found in North, Central, and South America (Eddleman et al. 1994). The subspecies of North and Central America are the California black rail and the eastern black rail (*L. j. jamaicensis*) (American Ornithological Union 1957). The California black rail was formerly known as the Farallon rail (*Creciscus coturniculus*), from a type specimen collected on the Farallon Islands of northern California in 1859 (Brewster 1907, Bent 1926).

Legal Status

The U.S. Fish & Wildlife Service lists the California black rail as a migratory nongame bird of special concern (USFWS 1995). In Arizona, it is listed as a wildlife species of special concern (WSC) (AGFD 2002). In California, this subspecies is listed as threatened (CDFG 2006). The California black rail is listed as endangered in Mexico (Hinojosa-Huerta 2004). It is not listed in Nevada as per Nevada Administrative Code 503.050.

Distribution

Historical Range

Bent (1926) lists the range of the California black rail as the Pacific coast of California. Grinnell (1915) stated that the black rail was a fairly common fall and winter visitor to the salt marshes around San Francisco Bay, the shores of San Mateo and Alameda counties, and Point Reyes Station at the head of Tomales Bay; interiorly to the Suisun marshes of Solano County and Stockton. Southerly, it was found at Santa Cruz, Hueneme in Ventura County, Orange in Orange County, Ballona in Los Angeles County, Riverside, and San Diego. The California black rail was a common summer resident and breeder in the salt marshes of San Diego Bay (Grinnell 1915). It was found nesting inland at Chino, San Bernardino County, in 1935 (Hanna 1935). It was also observed at Riverside in 1893 and San Bernardino in 1919, both in the month of August (Wall 1919). In Arizona, the black rail was listed as hypothetical (Swarth 1914, Phillips et al. 1964). The black rail was listed as a very rare migrant in the Tacoma, Washington area (Bowles 1906). It was also found inland in northern Baja California, Mexico (Huey 1928). There are no historical records for the black rail in Nevada or Utah (Hayward et al. 1976, Alcorn 1988).

In January 1947, a black rail was taken near Calipatria, Imperial County, adjacent to the Salton Sea (Laughlin 1947). The first observations of the black rail along the lower Colorado River were in June through August 1969. Four to six rails were observed near Imperial Dam (Snider 1969). Black rails were found in marshes adjacent to the Coachella Canal, east of the Salton Sea, in October 1974 and May 1975, as well as in marsh habitat around the Salton Sea in 1974 (McCaskie 1974, Jurek 1975). The first record for the black rail at the Bill Williams River delta was April 1978 and a population of 14 were found near Planet Ranch, on the Bill Williams River, in 2000 (Rosenberg et al. 1991, Conway et al. 2002). An estimated population of 30 California black rails were found during surveys along the All American Canal, west of Yuma, in 1984 (Kasprzyk et al. 1985). Individual black rails were observed at the Henderson Bird Viewing Preserve (HBVP) in 1999 and in September 2003 (Branca 2003). Piest and Campoy (1998) detected California black rails in the Cienega de Santa Clara, Sonora, Mexico. In 2000, California black rails were detected during surveys conducted in the Colorado River Delta, Baja California, and Sonora, Mexico (Hinojosa-Heurta et al. 2001).

Along the Virgin River, near Mesquite, Nevada, a couple of black rails were detected during July and August 2002, and one during July 2003 (Rathbun and Braden 2003, Braden et al. 2005). Black rails were also detected at several locations within the Grand Canyon, near Spencer Canyon, in 1998 and 1999 (Conway et al. 2002).

Current Range

During the breeding season, the California black rail is found north of San Francisco at Bodega Bay, Tomales Bay, and Bolinas Lagoon, the northern reaches of the San Francisco Bay estuary, and Morro Bay (Manolis1978). Further to the south, the rail is presently found at seep marshes and springs along the Coachella and All American canals, at one location on the New River south of the Salton Sea, and at Big Morongo Canyon in San Bernardino County (Conway et al. 2002). Along the lower Colorado River, the California black rail is found at the Cienega de Santa Clara and several other sites in the Colorado River Delta in Mexico (Hinojosa-Huerta et al. 2001). It is found from Laguna Dam north to Imperial Reservoir, portions of the Imperial NWR, on Colorado River Indian Tribe lands, at the Bill Williams River NWR, and the Havasu NWR (Conway et al. 2002, Corman 2005). Black rails found in the Salton Trough may be relict populations from marshes that persisted from ancient Lake Cahuilla (Evens et al. 1991).

Migration of the California black rail is poorly known (Eddleman et al. 1994). Adult populations appear to be sedentary, but juveniles may disperse erratically and colonize new areas (Huey 1916, Repking and Ohmart 1977, Todd 1977, Manolis1978, Flores and Eddleman 1991, Rosenberg et al. 1991). It is now a casual winter visitor along the coast of southern California, where it once bred (Garrett and Dunn 1981).

Populations within LCR MSCP Project Boundaries

At present, there are no known populations within reaches 1 and 2. In Reach 3, the Bill Williams River Delta is a known location; the Bill Williams River NWR lists the black rail as resident (Rosenberg et al. 1991, USFWS). In Reach 4, there appears to be no known populations, although an individual rail has been noted (Conway et al. 2002). Locations within reaches 5 and 6 contain the majority of California black rails within the LCR MSCP boundaries. In Reach 5, rails are found between Imperial Dam and Martinez Lake, at Ferguson Lake, and at managed wetlands on the Imperial NWR (Evans et al. 1991, USFWS 1994, Conway et al. 2002). In Reach 6, the California black rail is found on the California side of the Colorado River at West Pond and at Mittry Lake on the Arizona side of the river (Evans et al. 1991, Flores and Eddleman 1995, Conway et al. 2002). There are no known populations in Reach 7.

Life History

General Description

The black rail is the smallest rail in North America, with a total adult length of 10-15 cm, a wingspan of 22-28 cm, and a mean mass of 29-35 g. The California black rail is smaller and brighter colored than the eastern subspecies (Eddleman et al. 1994). The adult coloration is generally shades of pale to blackish gray, and the top of the head is darker than the surrounding plumage. The underparts are uniformly colored but lighter on the chin and throat. The undertail coverts and flanks are streaked with white and dark gray and washed with chestnut. The nape and upper back are also chestnut. The rest of the back, the uppertail coverts, and wing are shades of dark gray, sometimes with a chestnut or brown wash, and scattered with white spots. The amount of spotting varies between individuals. The tail feathers are brownish gray. The juvenile plumage is similar to the adult but duller, the white spots fewer and smaller, and the streaking on the flanks thinner and less distinct. The irides of the adult are shades of bright red, while juveniles' irides can range from olive to dull orange. The plumage of the California black rail is sexually dimorphic; the throat of the female is pale gray to white and the ventrum is medium to pale gray, whereas the male is darker with a pale to medium gray throat. The sexes are similar in size (Eddleman et al. 1994). The bill of the black rail is short and black (Sibley 2000). Length of the tarsus is approximately 2.5 cm and the color of the tarsi and toes is grayish brown (Flores and Eddleman 1991, Eddleman et at. 1994). The downy young are covered with black down and only distinguishable from other rails by their smaller size (Eddleman et al. 1994).

The primary call that is attributed to the male is described as *kickee-doo* or *kic-kic-kerr* and can be heard along the lower Colorado River throughout the year, although it is mostly heard between March and June (Repking 1975, Flores and Eddleman 1991, Conway et al. 2002, Corman 2005). The vocalization that is attributed to the female is described as *croo-croo-croo* or *who-who-who*, is rarely given, and is similar to the call of the least bittern (*Ixobrychus exilis*) and cuckoo (*Coccyzus* spp.) (Wayne 1905 *in* Bent 1926, Reynard 1974, Repking 1975, Flores and Eddleman 1991). Other vocalizations of the black rail heard along the lower Colorado River are *grrr* or *grr-grr-grr*, *churt* or *chirk*, a *yelp, ticuck,* and *tch-tch-tch* (Repking 1975, Repking and Ohmart 1977, Flores and Eddleman 1991, Conway 2005). Repking (1975) found the *churt* was principally heard in the winter and Flores (1991) stated that the call was given more during the non-nesting season than during nesting. California black rails vocalize mainly at twilight, especially at dusk, but are often heard during the daylight hours and rarely at night (Flores and Eddleman 1991).

The black rail is very secretive and seldom seen; it runs swiftly and quickly on the ground, is generally reluctant to fly, and its short flight is typical of rails (Eddleman et al. 1994). Its flight can be fast and strong over long distances (McMullen 1944 *in* Eddleman et al. 1994). It can also swim for short distances (Weske 1969).

The black rail loses its flight and tail feathers after breeding, usually between July 1 and August 31, and is flightless for up to 3 weeks (Flores 1991, Eddleman et al. 1994).

Seasons are described as early nesting (March-April), late nesting (May-July), post-nesting (August-October), and winter (November-February) (Flores and Eddleman 1995).

Breeding

There is little information available about pair formation (Eddleman et al. 1994). The California black rail may form pairs as early as late February, if the initiation of calling is an indication (Flores 1991).

The nest is a well-defined bowl, with a canopy of dead or living vegetation woven over the top and a ramp of dead vegetation leading from the substrate to an entrance on the side of the nest (Harlow 1913, Flores and Eddleman 1991). In one Arizona study, four out of five nests found were primarily made of southern cattail (*Typha domingensis*); the other was made of spikerush (E*leocharis* spp.). These nests were elevated above the mud substrate in clumps of vegetation: three in California or giant bulrush (*Scripus californicus*), one in southern cattail, and one in three-square bulrush (*S. americanus*) (Flores and Eddleman 1993). The dimensions of these nests ranged from 7 to 8 cm for inside diameter, 11 to 15 cm for outside diameter, and 3 to 7 cm for bowl depth. The height above the substrate ranged from 2 to 11 cm and the water depth at the nest site ranged from 0 to 3 cm (Flores and Eddleman 1993). These traits are similar to those for eastern black rail nests (Bent 1926, Flores and Eddleman 1993). Black rails have also been known to nest on top of a mat of dead vegetation from the previous years' growth (Pough 1951).

Nest records from the Western Foundation of Vertebrate Zoology indicate that the California black rail lays eggs between March 10 and July 6 (Eddleman et al. 1994). Five nests in Arizona were found between April 19 and July 23; the late date may indicate second nesting (Flores and Eddleman 1993). The clutch size at these nests ranged from 3 to 7 eggs; this is similar to clutch sizes reported from California (4-8 eggs) (Bent 1926). Both sexes incubate the eggs and the incubation period for the nests in Arizona ranged from17 to 20 days (Flores and Eddleman 1993).

Chicks apparently hatch one at a time and are born semiprecocial; they require brooding by one parent for the first few days after hatching (Heaton 1937 *in* Eddleman et al. 1994). Juvenile birds disperse widely from the breeding areas and may appear in atypical habitat (Eddleman et al. 1994).

Diet

Food habits of the black rail are poorly known (Flores and Eddleman 1991). Small aquatic and terrestrial invertebrates of less than 1 cm and seeds are main food items (Eddleman et al. 1994). Black rails in Arizona were found to consume predaceous diving beetles (Hydrophilidae), ground beetles (Carabidae), other beetles, earwigs (Dermaptera), and the seeds of Olney bulrush (*S. olneyi*), California bulrush, and southern cattail during the breeding season. In late summer and autumn, their diet included grasshoppers (Arcidae), beetles, ants (Formicidae), earwigs, spiders (Lycosidae), snails (Gastropoda), bulrush seeds, and insects (Reduvidae, Aphidae, Dolichopodidae, Kinnaridae, Homonoptera, and Diptera). During winter, they ate mostly bulrush seeds but also earwigs, beetles, ants, and cattail seeds (Flores and Eddleman 1991). The bill shape of the black rail suggests that it feeds by gleaning and pecking at individual items and relying on sight for finding food (Eddleman et al. 1994). The black rail is probably a daytime feeder and is active throughout the day (Weske 1969 *in* Eddleman et al. 1994, Flores 1991).

Habitat

The California black rail inhabits tidal marshes and freshwater marshes in the western United States and Mexico (Eddleman et al. 1994, Hinojosa-Huerta et al. 2001). It uses sites with shallower water than other North American rails (Eddleman et al. 1988). California black rail inhabit the drier portions of wetlands (Flores and Eddleman 1991). Inland sites, such as those along the lower Colorado River, are characterized by shallow, stable water level, gently sloping shorelines, and vegetation dominated by fine-stemmed bulrush (*Scirpus* spp.) or grasses (Repking and Ohmart 1977). Todd (1977) stated that black rails use dense stands of three-square bulrush along the lower Colorado River. Three-square bulrush is restricted to shallow water or moist soil (Conway et al. 2002). Flores (1991) described microhabitats of the black rail as having high stem densities and canopy coverage and being close to cover type edges.

Flores and Eddleman (1995) found that sites used by California black rails were based more on habitat structure than plant composition. The rails select areas with high stem densities and canopy coverage in shallow water, close to upland vegetation (Flores and Eddleman 1995). At Mittry Lake, California black rails chose marsh edges with a water depth less than 2.5 cm (1 in) that were dominated by giant or California bulrush and three-square bulrush (Flores and Eddleman 1995). The majority of sites where California black rails were detected in northern California were areas with water depths equal to or less than 3 cm (1.2 in) (Tecklin 1999 *in* Conway et al. 2002). The length of the tarsus of the black rail is roughly 2.5 cm and this may correspond with water depth at selected sites (Flores and Eddleman 1995). Black rails in Arizona were also found to use areas with southern cattail but only those with shallow water depths (Flores and Eddleman 1991). California black rails are also associated with plants of the upland/wetland interface, such as seep willow (*Baccharis glutinosa*), arrowweed (*Tessaria sericea*), saltgrass (*Distichlis spicata*), and cottonwood (*Populus fremontii*) (Conway et al. 2002). Flores and Eddleman (1995) detected few changes in seasonal use of vegetation types in their study at Mittry Lake, although rails used shrubs and three-square bulrush more during the post-breeding season than at other times, and juvenile rails selected inland saltgrass during the post-breeding season.

In Arizona, the home range size of the black rail was between 0.11 and 1.80 ha during the whole year. For all seasons, there were no significant differences in home range or core area sizes between the male and female. Generally, home range and core area sizes increased outside of the nesting season. While female-female, male-female, and juvenile-adult home ranges overlapped, home ranges of different males seldom overlapped, except during the winter (Flores 1991).

California black rails probably use only a fraction of the emergent vegetation within a wetland because of unsuitable water and structural conditions (Flores and Eddleman 1995).

Threats

Documented avian predators of the California black rail include great egret (*Casmerodius albus*), great blue heron (*Ardea herodias*), ring-billed gull (*Larus delawarensis*), northern harrier (*Circus cyaneus*), short-eared owl (*Asio flammeus*), and possibly loggerhead shrike (*Lanius ludovicianus*) (Huey 1926, Ewan 1928, Evans and Page 1986). A domestic cat captured a black rail in San Francisco, and rails have been collected after collisions with radio towers and buildings, as well as after encounters with automobiles (Gander 1930, Stoner 1945, Orr 1947). Rails forced from habitat by high tides are vulnerable to predation if they are unable to secure upland cover (Evans and Page 1986). This may be a possible scenario in managed wetlands along the lower Colorado River.

California black rails prefer marsh habitat with very shallow water levels of less than 3 cm (Flores and Eddleman 1991). Management actions that cause significant or long-term fluctuations of water level are a threat (Flores and Eddleman 1995). Seepage marshes are also threatened by the lining of canals (Jackson 1988). Another threat is the continued filling in or development of marshes along the lower Colorado River (Conway et al. 2002).

Selenium may be a threat to the California black rail along the lower Colorado River. Liver samples from four adult rails and one egg were analyzed for selenium. Selenium levels were elevated in two composites of two livers and in the egg. Similar tissue levels in mallards (*Anas platrhynchos*) can cause reproductive failure but implications for the black rail are unknown (Eddleman et al. 1994).

Adult black rails lose their flight and tail feathers during their definite prebasic molt between July 1 and August 31; they remain flightless for up to 3 weeks (Eddleman et al. 1994). Wildfires or controlled fires during this time period are a potential threat.

LCR MSCP Conservation Measures

BLRA1 — Create 130 acres of California black rail habitat.
Of the 512 acres (209 ha) of LCR MSCP-created marsh, 130 acres (52.5 ha) will be created and managed to provide California black rail habitat near occupied habitat in reaches 5 and 6. This habitat will be provided by designing and managing at least 130 acres (52.5 ha) of the 512 acres (209 ha) of created Yuma clapper rail (*Rallus longirostris yumanensis*) habitat to provide habitat for both species. Habitat will be created in patches as large as possible but will not be created in patches smaller than 5 acres (2.0 ha). Additional California black rail habitat will be provided by marsh vegetation that becomes established along margins of the 360 acres (145 ha) of backwaters that will be created in reaches 5 and 6. These small patches of habitat provide cover for dispersing rails, thereby facilitating linkages between existing breeding populations and the colonization of created habitats (LCR MSCP 2004).

BLRA2 — Maintain existing important California black rail habitat areas.
The Applicants, under agreements with cooperating land management agencies, will provide funding to those agencies to maintain a portion of existing California black rail habitat in the LCR MSCP planning area. Maintaining important existing habitat areas is necessary to ensure the continued existence of California black rails in the LCR MSCP planning area, provide for the production of individuals that could disperse to and nest in LCR MSCP-created habitats, and support future recovery of the species. Habitat maintenance would likely be undertaken in conjunction with the maintenance of existing Yuma clapper rail habitat (LCR MSCP 2004).

LCR MSCP Research and Monitoring Needs

California black rail populations are restricted along the LCR. Information on distribution, dispersal, population trends, and habitat requirements are not fully understood. To successfully create and maintain black rail habitat, these important factors needed to be undetermined.

There is a need to evaluate black rail distribution throughout the LCR MSCP area. Black rails have been recently encountered in Reach 1 (the Grand Canyon) and surrounding areas, such as the HBVP and Mesquite (Conway et al. 2002, Branca 2003, Rathbun and Braden 2003, Braden et al. 2005).

Conservation Measure BLRA1 requires California black rail habitat creation projects be conducted within reaches 5 and 6, near occupied habitat. Juvenile black rails may disperse widely from breeding areas (Eddleman et al. 1994). Black rail demographics, especially for populations found within reaches 5 and 6, need to be determined, including dispersal distances, population trends, and habitat requirements. These data will be used to design habitat mosaics for black rails, Yuma clapper rails, and least bitterns. Dispersal data are important for site selection in planning future habitat creation projects within reaches 5 and 6.

California black rails and egg had elevated selenium levels at Mittry Lake, although eggs from five monitored nests successfully hatched (Flores and Eddleman 1991). The effects of varying selenium levels on reproduction and survival for black rails should be evaluated to determine if selenium bio-accumulation has a detrimental affect on black rail populations within the LCR MSCP planning area.

Literature Cited

Alcorn, J.R. 1988. The birds of Nevada. Fairview West Publishing, Fallon, Nevada. 418 p.

American Ornithologist's Union. 1957. Checklist of North American birds, 5th edition. American Ornithologist's Union, Baltimore, Washington, DC.

Arizona Game and Fish Department. 2002. *Laterallus jamaicensis coturniculus*. Unpublished abstract compiled and edited by the Heritage Data Management System, Arizona Game and Fish Department, Phoenix, Arizona. 5 p.

Bent, A.C. 1926. Life histories of North American marsh birds. U.S. National Museum Bulletin No. 135. Dover Publications, New York, NY. 392 p.

Bowles, J.H. 1906. A list of the birds of Tacoma, Washington and vicinity. Auk 23:138-148.

Braden, G.T., K. Carter, and M.R. Rathbun. 2005. The status of Yuma clapper rail and yellow-billed cuckoo along portions of the Virgin River and Muddy River in southern Nevada: 2003, Final. Report for Southern Nevada Water Authority, Las Vegas, NV. 33 p.

Branca, J. 2003. Henderson bird viewing preserve notes. http://list.audubon.org/archives/nvbirds.html.

Brewster, W. 1907. Notes on the black rail of California. Condor 11:47-49.

California Department of Fish and Game. 2006. State and Federally listed endangered and threatened animals of California. California Department of Fish and Game, Sacramento, California. 12 p.

Conway, C.J., C. Sulzman, and B. Raulston. 2002. Population trends, distribution, and monitoring protocols for the California black rail. Technical report (Heritage Program IIPAM Grant # 199010). Arizona Game and Fish Department, Phoenix, Arizona.

Conway, C.J. 2005. Standardized North American marsh bird monitoring protocols. Wildlife Research Report #2005-04. U.S. Geological Survey, Arizona Cooperative Fish and Wildlife Research Unit, Tucson, Arizona.

Corman, T.E. 2005. Black Rail (*Laterallus jamaicensis*). *In:* T.E. Corman and C. Wise-Gervais (editors), Arizona Breeding Bird Atlas University of New Mexico Press, Albuquerque, New Mexico. 636 p.

Eddleman, W.R., F.L. Knopf, B. Meanly, F.A. Reid, and R. Zembal. 1988. Conservation of North American rallids. Wilson Bulletin 100:458-475.

Eddleman, W.R., R.E. Flores, and M.L. Legare. 1994. Black Rail (*Laterallus jamaicensis*). *In:* A. Poole and F. Gill (editors), The Birds of North America, No. 123. Philadelphia: The Academy of Natural Sciences, Washington, DC: The American Ornithologist's Union.

Evens, J., and G.W. Page. 1986. Predation on black rails during high tides in salt marshes. Condor 88:107-109.

Evens, J.G., G.W. Page, S.A. Laymon, and R. Stallcup. Distribution, relative abundance, and status of the California black rail in western North America. Condor 93:952-966.

Ewan, J. 1928. California black rail in Los Angeles County. Condor 30:247.

Flores, R.E. 1991. Ecology of the California black rail in southwestern Arizona. Master's Thesis, University of Rhode Island, Kingston.

Flores, R.E. 1993. Nesting biology of the California black rail in southwestern Arizona. Western Birds 24:81-88.

Flores, R.E. 1995. California black rail use of habitat in southwestern Arizona. Journal of Wildlife Management 59:357-363.

Flores, R.E., and W.R. Eddleman. 1991. Ecology of the California black rail in southwestern Arizona. Final Report, Bureau of Reclamation, Yuma Project Office and Arizona Department of Game and Fish, Yuma, Arizona.

Gander, F.F. 1930. A black rail leaves the salt marsh. Condor 32:211.

Garrett, K., and J. Dunn. 1981. Birds of southern California: Status and distribution. Los Angeles Audubon Society, Los Angeles, California. 408 p.

Grinnell, J. 1915. A distributional list of the birds of California. Cooper Ornithological Club, Pacific Coast Avifauna, No.11. The Club, Hollywood. 217 p.

Hanna, W.C. 1935. Farallon rail nesting inland. Condor 37:81-82.

Harlow, R.C. 1913. Nesting of the black rail (*Creciscus jamaicensis*) in New Jersey. Auk 30:269.

Hayward, C.L., C. Cottam, A.M. Woodbury, and H.E. Frost. 1976. Birds of Utah. Great Basin Naturalist Memoirs. Brigham Young University Press, Provo, Utah. 229 p.

Heaton, H.L. 1937. Baby Farallon rails. Oologist 54:102-103.

Hinojosa-Huerta, O., W.W. Shaw, and S. DeStefano. 2001. Detections of California black rail in the Colorado River Delta, Mexico. Western Birds 32:229-232.

Hinojosa-Huerta, O., H. Iturribarría-Rojas, Y. Carrillo-Guerrero, M. de la Garza-Treviño, and E. Zamora-Hernández. 2004. Bird conservation plan for the Colorado River Delta. Pronatura Noroeste, Dirección Sonora. San Luis Río Colorado, Sonora, México.

Huey, L.M. 1916. The Farallon rails of San Diego County. Condor 18:58-62.

Huey, L.M.. 1926. Bats eaten by short-eared owl. Auk 43:96-97.

Huey, L.M. 1928. Some bird records from northern Lower California. Condor 30:158-159.

Jackson, J. 1988. Surveys for the Yuma clapper rail (*Rallus longirostris yumanensis*) and California black rail (*Laterallus jamaicensis coturniculus*) along the 38-mile unlined portion of the Coachella Canal and adjacent wetlands, Imperial and Riverside counties, California. *In:* Coachella Canal Lining Project, Draft Environmental Impact Statement, Environmental Impact Report, Environmental Appendix, December 1993. USDI, Bureau of Reclamation, Denver Office, Ecology Branch, Denver, Colorado.

Jurek, R.M. 1975. Survey of Yuma clapper rails and California black rails along Coachella Canal, Imperial County, May 1975. California Department of Fish and Game, Nongame Wildlife Investigations Report, Project W-54-R7, Job 1-3. 6 p.

Kasprzyk, M.R., R. Smith, and R. Bransfield. 1987. Avian species composition and densities of a wetland community along the All-American Canal, Imperial County, California. *In:* All-American Canal relocation study, California, Environmental Appendix, U.S. Bureau of Reclamation, Boulder City, Nevada. 11 p.

Laughlin, J. 1947. Black rail at Salton Sea, California. Condor 49:132.

Lower Colorado River Multi-Species Conservation Program. 2004. Lower Colorado River Multi-Species Program, Volume II: Habitat Conservation Plan. Final. December 17. (J&S 00450.00) Sacramento, California.

Manolis, T. 1978. Status of the black rail in central California. Western Birds 9:151-158.

McCaskie, G. 1974. Southern Pacific coast region. American Birds 28:949.

McMullen, T.E. 1944. The black rail. Cassinia 34:20-21.

Orr, R.T. 1947. Occurrence of black rail in San Francisco. Condor 49:41.

Piest, L., and J. Campoy. 1998. Report of Yuma clapper rail surveys at Cienega de Santa Clara, Sonora, 1998. Unpublished report. Arizona Game and Fish Department. 9 p.

Phillips, A., J. Marshall, and G. Monson. 1964. The birds of Arizona. University of Arizona Press, Tucson, Arizona. 212 p.

Pough, R.H. 1951. Audubon waterbird guide. Doubleday Co. Garden City, New York. 352 p.

Rathbun, M.R., G.T. Braden. 2003. The status of Yuma clapper rail and yellow-billed cuckoo along portions of the Virgin River and Muddy River in southern Nevada for 2002. Report to Southern Nevada Water Authority, Las Vegas, Nevada. 29 p.

Repking, C.F. 1975. Distribution and habitat requirements of the black rail (*Laterallus Jamaicensis*) along the lower Colorado River. Master's Thesis. Arizona State University, Tempe.

Repking, C.F., and R.D. Ohmart. 1977. Distribution and density of black rail populations along the lower Colorado River. Condor 79:186-189.

Reynard, G.B. 1974. Some vocalizations of the black, yellow, and Virginia rails. Auk 91:747-756.

Rosenberg, K.V., R.D. Ohmart, W.C. Hunter, and B.W. Anderson. 1991. Birds of the lower Colorado River Valley. University of Arizona Press, Tucson. 416 p.

Sibley, D.A. 2000. The Sibley Guide to Birds. The National Audubon Society. Alfred A. Knopf, New York. 544 p.

Snider, P.R. 1969. Southwest region. Audubon Field Notes. 23:681.

Stoner, E.A. 1945. The black rail at Benicia, California. Condor 47:81.

Swarth, H.S. 1914. A distributional list of the birds of Arizona. Cooper Ornithological Club, Pacific Coast Avifauna, No. 10. The Club, Hollywood. 133 p.

Todd, R.L. 1977. Black rail, little black rail, black crake, Farallon rail (*Laterallus jamaicensis*). Pages 71-83 *In:* G. C. Sanderson (editors), Management of migratory shore and upland game birds in North America. Intl. Assoc. Fish and Wildl. Agen., Washington, DC.

U.S. Fish and Wildlife Service. No date. Birds of the Bill Williams River National Wildlife Refuge. U.S. Fish and Wildlife Service. Unpaginated. Jamestown, ND: Northern Prairie Wildlife Research Center Online. http://www.npwrc.usgs.govbillwill.htm (Version 22MAY98).

U.S. Fish and Wildlife Service. 1994. Birds of the Imperial National Wildlife Refuge. Unpaginated. Jamestown, ND: Northern Prairie Wildlife Research Center Online. http://npwrc.usgs.govimperial.htm (Version 22MAY98).

U.S. Fish and Wildlife Service. 1995. Migratory nongame birds of special concern in the United States: The 1995 list. Office of Migratory Bird Management, Washington, DC. 28 p.

Wall, E. 1919. California black rail at San Bernardino, California. Condor 21:238.

Weske, J.S. 1969. An ecological study of the black rail in Dorchester County, Maryland. Master's Thesis, Cornell University, Ithaca, NY.

YELLOW-BILLED CUCKOO
(*Coccyzus americanus occidentalis*)

Legal Status

Federal Listing

From 1972 to 1981, the yellow-billed cuckoo was blue listed by the National Audubon Society. In 1982, the cuckoo became a species of special concern. In 1986, the western yellow-billed cuckoo was petitioned as endangered in the states of California, Washington, Oregon, Idaho, and Nevada (Manolis et al. 1986). The 12-month finding stated that the petitioned action was not warranted, finding that the petitioned area did not encompass either a distinct subspecies or a distinct population segment. The finding cited: 1) a study of geographic variation in the species that concluded the morphological differences between eastern and western birds were too small to merit separate subspecies (Banks 1988), and 2) that the petitioned area did not encompass a distinct population segment. In 1998, 22 groups filed a petition with the U.S. Fish and Wildlife Service seeking endangered species status for the western subspecies. This petition was filed due to the decline or local extirpation of the yellow-billed cuckoo throughout the western portion of its range in the past 60 years. In 2000, the USFWS announced that the petition presented substantial scientific information to indicate that the listing of the cuckoo may be warranted. In 2001, the USFWS decided that the western birds represent a distinct population segment (DPS), and are now a candidate for Federal endangered species status (USFWS 2001). The western yellow-billed cuckoo DPS is considered threatened but precluded by higher priority listing actions, and currently sits on the candidate species list as of July 18, 2001. The U.S. Forest Service considers the cuckoo a Region 2 Sensitive species.

State Listings

In California, the western yellow-billed cuckoo became listed as threatened in 1971. In 1987, the species became listed as a Status 1 species (Critically Imperiled, Endangered) by the California Department of Fish and Game. In 1988, the western yellow-billed cuckoo became listed as threatened by the Arizona Game and Fish Department. The western yellow-billed cuckoo is considered a threatened species in Utah. In Nevada, the species is listed as critically imperiled and is proposed for protection as threatened. Currently, it is listed as endangered by the Nevada Natural Heritage Program.

Distribution

Historical Range

Historically, the western yellow-billed cuckoos occupied and bred in the formerly extensive riparian zone from southwest British Columbia, Washington, Oregon, southwestern Idaho, California, Nevada, northern Utah, central and western Colorado, Arizona, New Mexico, and western Texas, and south and west to southern Baja

California, Sinaloa, and Chihuahua in Mexico (Hughes 1999). Like many riparian obligate species, the breeding distribution and number of western yellow-billed cuckoos has declined in the past 80 years throughout western North America (Gaines and Laymon 1984, Corman and Magill 2000). The initial decline was most likely linked to the extensive loss of riparian habitat within the western breeding range of the species. During the late 1800s and early 1900s, large areas of virtually continuous riparian habitat in the western United States were destroyed by human activities, including conversion to agriculture, submersion under reservoirs, and channelization for flood control. Along with local declines, there was an overall range contraction, retracting from British Columbia in the 1920s, Washington in the 1930s, Oregon in the 1940s, and in northern California in the 1950s (Laymon and Halterman 1987).

From the early part of the 20th century, yellow-billed cuckoos were frequently listed as a fairly common to common breeding species within the extensive riparian forests dominated by cottonwood, willow, and/or mesquite throughout Arizona and California (Gaines 1974, Groschupf 1987). In California, Grinnell (1915) described this species as a common breeder, but by 1940, the cuckoo was much reduced in population due to the removal of widely essential habitat conditions (Grinnell and Miller 1944). In Arizona, often-dense woodlands once extended for many kilometers along the lower Colorado, Gila, Salt, Verde, Santa Cruz and San Pedro river valleys (Corman and Magill 2000). On the lower Colorado River, Grinnell and Miller (1944) cite only Stephen’s (1903) observation of several cuckoos near Needles in 1902. The paucity of historical data probably reflects an absence of observers during the breeding season (Gaines and Laymon 1984). Until very recently, yellow-billed cuckoos were a fairly common but local summer breeder from June through August of the bottomland willow-cottonwood habitat along the lower Colorado (Rosenberg et al. 1991). A summary of Arizona and California cuckoo surveys indicates a drastic decline in cuckoo numbers since the 1970s. The yellow-billed cuckoo was once considered a common nester in Arizona river bottoms; however, severe declines have occurred statewide. A statewide Arizona survey of suitable habitat, conducted in 1977, found an estimated total of 205-214 pairs, with more than half of these detected along the lower Colorado River (Gaines and Laymon 1984). Gaines (1974), using species-specific protocols, found 141 birds in California, with 65 cuckoos found along the lower Colorado River. The Bill Williams Delta has historically had the most concentrated western population, with 57 pairs detected in 1977 (Rosenberg et al. 1991). Estimates suggested less than 200 pairs remained in Arizona by 1986 (Laymon and Halterman 1987), and less than 50 pairs 5 years later (Ehrlich at al. 1992). A detailed chronological history of the western yellow-billed cuckoo in Arizona and California follows below.

Hamilton and Hamilton (1965) surveyed for cuckoos in mid-June 1964, near Laguna Dam on the Colorado River. Cuckoo observations suggested that the density of yellow-billed cuckoos was similar to the San Pedro River in southeastern Arizona, as evidenced from the frequent songs, even suggesting that the population was decidedly denser. A substantial population of yellow-billed cuckoos was found on the Lower Colorado River, north of Laguna Dam, during the 1960s and 1970s (Gaines and Laymon 1984). From 1964 to 1975, 4 to 12 cuckoos per season were reported near Laguna Dam in June and

July (Gaines and Laymon 1984). In 1977, Laguna Dam had at least three pairs of cuckoos occupying a 12-ha site that was approximately 40% willow (Gaines and Laymon 1984). Tamarisk dominated this habitat when it was surveyed in 1986, with no cuckoos present on the surveys (Laymon and Halterman 1987).

Surveys in 1983 found 65 cuckoos at 16 sites scattered along the lower Colorado River from Needles, California, to Laguna Dam. Of the 51 cuckoos sighted, 79% were perched in willows, 11% in mesquite, 8% in cottonwood, and 2% in tamarisk. Occupied yellow-billed cuckoo habitat near the Topock marsh area consisted of a large expanse of arrowweed, tamarisk, willow, and mesquite 3-4 m high. Willows 7-9 m tall formed an open overstory. Canopy cover varied from 10 to 20%, with understory cover from 80 to 90%. At least three pairs were found in a 12-ha site near Laguna Dam, the highest cuckoo density found during this survey on the Colorado River mainstem. Twenty-one cuckoos were found in 120 ha of dense willows on Imperial National Wildlife Refuge and in Picacho State Park. The canopy varied from 4 to 10 m and canopy cover was 80 to 90%, with willow groves forming 70 m strips along the river channel. The forests on the Bill Williams floodplain supported the highest density of cuckoos. Surveyors detected 11, including 8 in 12 ha of willows and cottonwoods. Canopy height measured 17 m and canopy cover measured 80%. In general, sites inhabited by cuckoos were characterized by at least 20% willow cover, dense shrub and understory foliage, and open water or marsh within 100 m. There was no evidence that suitable habitat was not being utilized, suggesting that habitat availability was limiting this cuckoo population on the lower Colorado River (Gaines and Laymon 1984). This survey supported previous estimates of cuckoo abundance in 1975 and 1976. Rosenberg et al. 1991 estimated 244 cuckoos on the Colorado River from Davis Dam to Morelos Dam, Mexico, and 114 cuckoos on the Bill Williams River Delta.

Groschupf (1987) estimated 846 pairs of yellow-billed cuckoo in Arizona along the lower Colorado, Gila, Salt, Verde, San Pedro, and the Santa Cruz rivers. These estimates were based on extrapolations that assumed cuckoo densities to be constant throughout the habitat, and cuckoos occupied all or most of the available suitable habitat. Laymon and Halterman (1987) estimated cuckoo populations in Arizona to be fewer than 200 pairs, based on previous knowledge of habitat use by the species and the existing suitable breeding habitat (Corman and Magill 2000).

Current Range

By 1998, the western yellow-billed cuckoo was known to breed in California, Arizona, New Mexico, extreme western Texas, Sonora, Chihuahua, and irregularly to Zacatecas, Mexico (Hughes 1999). The subspecies is a probable local, but irregular, breeder in Utah, Nevada, and western Colorado. Known populations in the west include areas along the Sacramento and Kern rivers in California (Laymon 2000); the Verde, San Pedro, Santa Cruz, Bill Williams, and Gila rivers, and Cienega and Sonoita creeks in Arizona; and the lower Colorado River (Corman and Magill 2000).

The first statewide survey conducted along 25 drainages in Arizona from 1998 to 1999 found 514-516 yellow-billed cuckoos, thought to represent 206 to 207 pairs (Corman and

Magill 2000). The largest concentrated population of cuckoos in the western United States occurs on the San Pedro River (Halterman 2002). The San Pedro River National Conservation Area encompasses 58,000 acres of public land in Cochise County, Arizona, between the Mexican border and St. David, Arizona. The San Pedro is an extensive riparian corridor, dominated by Fremont cottonwood and Goodding's willow, mixed with Arizona ash, walnut, and netleaf hackberry, and flanked by extensive mesquite 6-7 m high. In 2005, four surveys had a total of 163 detections, estimated at 96 individuals, down from 135 individuals in 2004 and 165 in 2003. There seems to be some evidence of a declining trend in this population (Halterman 2006).

Three river drainages were surveyed behind Alamo dam: the Bill Williams River at Brown's Crossing, the Santa Maria River, and the Big Sandy River. In 1998, the Arizona Game and Fish Department conducted surveys and found a total of seven pairs at these sites (Halterman 2001). In 2000, Halterman detected a total of seven adults on drainages behind Alamo Dam. Habitat consists predominantly of tamarisk, with willow and cottonwood mixed in.

Surveys on the Sacramento River, from 1987 to 1990, estimated a fluctuating population of 23 to 35 pairs (Halterman 1991). Continuous surveys on the South Fork of the Kern River, from 1985 to 1996, estimated a population that varied from a low of two pairs in 1990 to a high of 24 pairs in 1992 (Laymon et al. 1997). The Audubon California's Kern River Preserve and U.S. Fish and Wildlife Service South Fork Wildlife Area contain 2000 acres of cottonwood-willow habitat, the state's largest lowland riparian forest. In 2004, surveyors detected 44 cuckoos, representing 28 individuals. In 2003, surveyors detected 40 cuckoos, representing 28 individuals. The population on the Kern appears to be fairly stable.

In southern Nevada, Nevada Division of Wildlife (NDOW) coordinated surveys on Warm Springs Ranch in Moapa Valley, north Pahranagat Valley, Oasis Valley south, Pahranagat Valley—Crystal South, Beaver Dam, and Meadow Valley Wash from river miles 39 to 44. In 2000, NDOW detected 19 individuals, representing four pairs and 11 single cuckoos, at Warm Springs Ranch (Gallager et al. 2001). The Moapa Valley has warm springs that flow into the Muddy River. These waterways were lined with mature riparian woodland consisting of Fremont cottonwood, velvet ash, Washington fan palm, and Goodding's willow. The riparian area used by cuckoos at Warm Springs covers 30 ha, consisting of narrow stringers along the Muddy River, tributary streams, and old irrigation ditches, none greater than 40 m wide. Southern Sierra Research Station (SSRS) surveyed four sites in southern Nevada: Clover Creek at Caliente, Upper Pahranagat Lake, Virgin River at Littlefield, and Meadow Valley Wash from river miles 39 to 57. In 2000, SSRS detected one mated cuckoo at Pahranagat and one individual at Littlefield (Halterman 2001). In 2001, SSRS detected four pairs and five single cuckoos. In 2002, SSRS and NDOW detected one to two pairs (Halterman 2003).

Populations within LCR MSCP Project Boundary

Prior to 1998, yellow-billed cuckoos were detected on the lower Colorado River at Lake Mead Delta, Topock Marsh, Topock Gorge, Bill Williams NWR, south of Ehrenberg, Clark Ranch, north of Blythe, Cibola NWR, Paradise Valley, Adobe Lake, McAllister Lake, Imperial NWR, Picacho State Recreation Area, Martinez Lake, below Imperial Dam, Laguna Dam, Gila River confluence, Morelos Dam, Hunter's Hole, and Gadsen Bend (Corman and Magill 2000, Gaines and Laymon 1984, Rosenberg et al. 1991).

The Arizona Breeding Bird Atlas (1993-2000) confirmed breeding cuckoos on the Bill Williams River, with possible or probable breeding documented at 12 other sites along the lower Colorado River (Corman 2005). In 1999, Corman and Magill (2000) detected eight pairs and eight single cuckoos along the lower Colorado River in native riparian habitat patches from Cibola National Wildlife Refuge south to Gadsen Bend. Sites included Gadsen Bend, Gila River Confluence, Hart Mine Wash, Imperial National Wildlife Refuge, Indian Wash, Island Reveg, McAllister Lake, McAllister Wash, Mittry Lake, and Morelos Dam.

SWCA Environmental Consultants and San Bernardino County Museum have conducted southwestern willow flycatcher studies on the lower Colorado River from 1997 to 2005. Surveys have confirmed breeding yellow-billed cuckoos on the following study sites: Virgin River, Pahranagat, Grand Canyon, Bill Williams, and Ehrenberg. Cuckoos have been present at least two years at the following sites: Virgin River, Pahranagat, Grand Canyon, Topock Marsh, Lake Havasu, Bill Williams, Ehrenberg, Walker Lake, Paradise Valley, Adobe Lake, Taylor Lake, Gila Confluence, and Hunter's Hole (Yuma) (Koronkiewicz et al. 2004, 2005, 2006, McKernan and Braden 2002).

The Bill Williams River National Wildlife Refuge supports the largest population of yellow-billed cuckoos within the LCR MSCP boundary. In 1998, the Bill Williams River population represented the second largest cuckoo population in Arizona. The Bill Williams River National Wildlife Refuge consists of riparian habitat along the Bill Williams River from Lake Havasu upstream to Planet Ranch, approximately 16 km (10 miles). The Bill Williams cuckoo population was monitored in 1993, 1994, and 1997-2004. In 2004, surveyors made 35 detections, representing an estimated 22 cuckoos, in 2003, surveyors mad 80 detections, estimated at 42 individuals, in 2002, surveyors made 34 detections, estimated at 8 pairs, and in 2001, surveyors made 90 detections, estimated at 28-30 pairs and 20-35 single cuckoos. Prior to 2001, reports estimated pairs rather than individual detections. Estimates ranged from 6-9 pairs in 1999 to 28-30 pairs in 1993.

Life History

General description

Coccyzus americanus occidentalis (western) formerly bred from southwestern British Columbia, western Washington, northern Utah, central Colorado, and western Texas and west to southern Baja California, Sinaloa, and Chihuahua in Mexico. *C. americanus americanus* (eastern) occupies the remainder of the range in eastern North America, including eastern Mexico and the Greater Antilles. The boundary between the subspecies is considered to be the Pecos River in Texas and New Mexico.

Two subspecies are recognized, although the validity of these has been debated (Hughes 1999). Ridgeway (1887) considered cuckoos from western North America a separate subspecies because of the slightly greater total length and larger, stouter bill. Multiple authors recognize distinct subspecies, including Ridgeway (1887), Franzreb and Laymon (1993), and Pruett et al. (2001), while others disagree (Banks 1988, Fleischer 2001). The taxonomic status of the subspecies currently remains tentative (Corman and Magill 2000). Data collected in eastern and western cuckoos found significant differences in wing, tail, and bill lengths, and in bill depth, and also outlined potential behavioral, vocal, and ecological differences. Two subspecies should be retained pending further examination (Hughes 1999).

The yellow-billed cuckoo is slender, long-tailed (length 26-30 cm, mass 55-65 g) and zygodactylous: two inner toes point forward and two outer toes are reversed. The head and upper parts are plain grayish brown, faintly glossed with olive. Underparts are dull white, faintly shaded with pale bluish gray or pale buff. It has a long, graduated tail (about 15 cm), plain grayish brown above and black below. Outer rectrices are broadly tipped with bright white, giving the appearance of 6 large, white spots on the underside. Distinctive tail pattern is noticeable both in flight and when perched. Wings are plain grayish brown above. Outer webs of primaries are dull cinnamon-rufous tipped with brown, and large rufous wing patches are visible during flight. The moderately long, curved bill has a hooked tip, the upper mandible is black, and the lower mandible is yellow to orange yellow at the base with a dark tip. The legs are blue-gray (Halterman 1991). Cuckoos are sexually and seasonally monomorphic in plumage. Juveniles generally resemble adults, but have wing coverts tinged with cinnamon brown and a less distinct undertail pattern (Hughes 1999). Second-year birds have a yellow orbital skin (Pyle 1997).

Migration

Yellow-billed cuckoos are long-distance migrants. Hamilton and Hamilton (1965) postulate a nomadic phase prior to breeding during which cuckoos appraise local food resources before establishing territories. Great numbers are recruited to areas of high caterpillar density during outbreaks (Hamilton and Hamilton 1965, Nolan and Thompson 1975).

The western yellow-billed cuckoo arrives on the breeding grounds beginning in mid- to late May, which is 4 to 8 weeks later than eastern cuckoos occurring at the same latitude (Franzreb and Laymon 1993). Western populations breed in June to August, with a peak occurring in mid-July to early August. The earliest recorded spring arrival date for California is 23 April, the only April record for the state. Although a few individuals arrive in May, the majority of breeding pairs arrive in June and some breeders may arrive as late as early July (Gaines and Laymon 1984). Yellow-billed cuckoos were rarely detected in spring migration in California away from the breeding grounds. In Arizona, yellow-billed cuckoos are the latest spring migrants to arrive on the breeding grounds. A few individuals arrive in mid- to late May, but the majority do not arrive until mid-June, with late migrants occasionally straggling into early July (Corman 2005). In Arizona, the extreme egg dates reported for the cuckoo are 15 June to 24 August (Corman and Magill

2000, Groschupf 1987). Cuckoos arrive in Nevada in late May to early June (Hughes 1999). Peak nesting activity typically occurs from July to early August, and continues through August, frequently into September, with the latest nesting observed on 18 September (Corman 2005). Yellow-billed cuckoos depart the breeding grounds between late July and mid-September.

Cuckoos are the last migratory summer breeder to arrive in the lower Colorado River Valley, with some individuals still arriving in mid- to late June and lingering through early September. Nesting commences in early July and continues through August. Territories are quickly established, young are raised, and individuals depart by the end of August, spending the shortest amount of time of any breeding species in the Colorado River Valley (Rosenberg et al. 1991).

Diet

Yellow-billed cuckoos usually glean prey items from foliage or branches, sometimes while hovering, or sallying from a perch to capture prey on the wing (Elhrich et al. 1992). Yellow-billed cuckoos feed primarily on slow-moving insects, including Orthoptera (grasshoppers, crickets, katydids), Lepidoptera (primarily caterpillars), and various bugs (Hemiptera) and beetles (Coleoptera). Beal (1898) studied yellow-billed cuckoo stomach contents from across the range and found a relatively uniform diet consisting of caterpillars (49%), Orthoptera (30%), and various other insects (18%). In a sample of stomach contents from Nebraska, Bent (1940) found that 73% of the total prey mass was made up of Orthoptera. Larvae of the family Sphingidae (sphinx moths, Lepidoptera) have been noted as an important food source for yellow-billed cuckoos, and the lack of such prey has been implicated in the decline of the western subspecies.

A detailed study of food items brought to nests on the Kern River, California, found 45% green caterpillars (primarily sphinx moth larvae), 24% tree frogs, 22% katydids, and 9% grasshoppers. A sample of 2,420 prey items being fed to young cuckoos were identified at 30 nests at the South Fork of the Kern River. The provisions that are brought to the young are whole prey items, with the exception of during the first few hours after hatching when young are fed regurgitated food (S.A. Laymon, pers. obs. in Ehrilch et al. 1988). The number of eggs laid was positively correlated to the percent of katydids fed to the young ($r^2 = 0.55$, $p = 0.04$) and negatively correlated to the percent of green caterpillars fed to the young ($r^2 = 0.51$, $p = 0.05$). Total number of young fledged per pair was correlated to the capture time of caterpillars, katydids, and all food types, with shorter capture time correlated to more young fledged (Laymon et al. 1997). The caterpillars and katydids appear to be the preferred food, while the tree frogs and grasshoppers appear to be "fast food" that can be caught quickly to placate the young while the adults then go after the preferred food. Food resources vary greatly from year to year and have a significant impact on reproductive success (Laymon et al. 1997).

In 2000, cicadas and katydids were the preferred food on the Bill Williams NWR, accounting for 53% of food items, and were delivered at a rate of 0.7 to 1.3 food items to nest per hour (Halterman 2001). In 1993, the diet of the young surveyed at the Bill Williams NWR consisted of cicadas (39.2%), katydids (17.7%), green caterpillars and sphinx moth larvae (14.5%), grasshoppers (7.6%), mantids (4.4%), and other (16.4%) (Laymon and Halterman 1994).

Foraging Strategy

Yellow-billed cuckoos are primarily foliage gleaners, although at times they sally from a perch and catch flying prey, such as dragonflies and butterflies, or drop to the ground to catch grasshoppers or tree frogs. Two foraging strategies are employed by the yellow-billed cuckoo when they are foliage gleaning. The primarily strategy is to hop slowly from location to location, sitting for several minutes at each location, watching for motion of their primarily green prey on a green leaf background. An alternate strategy, used less much frequently, is to dive into the foliage of a likely looking spot in hopes of dislodging prey (Laymon 1998).

While nests are almost always placed in willows, cottonwoods are extremely important for foraging. Two male cuckoos at the South Fork of the Kern River, equipped with radio transmitters, foraged much more in cottonwoods than would have been predicted by the cottonwood's abundance within the cuckoos' home range (Laymon and Halterman 1985). At the South Fork of the Kern River, cuckoos are found more often at upland sites early in the season in wet years, but not in dry years. It is likely that flooding in wet years reduces the survival of the larvae of the preferred prey (katydid and sphinx moth), which winter underground (Laymon 1998). This forces the cuckoos to forage in upland areas that were not flooded until the prey base in the lower floodplain begins to recover later in the breeding season. The fact that most extant riparian habitat is in the primary floodplain could cause a large reduction in the prey base and be a major cause of the decline of cuckoos in the western Unites States. Restoration efforts should consider planting at least a portion of forests on upper terrace sites that do not regularly flood, or eliminating watering of mature stands during the winter.

Vocalizations

The distinctive stuttering call of the yellow-billed cuckoo is heard much more often than the bird is actually seen (Corman 2005). Cuckoos give three main types of calls at irregular intervals: contact calls, comprising a series of mixed ‘kuks’ and ‘kowlps’; cooing or cawing calls; and several alarm calls (Halterman et al. 2002).
Cuckoo vocalizations are described in detail by Hughes (1999) and others (Bent 1940, Hamilton and Hamilton 1965, Potter 1980).

Several researchers have hypothesized sexual dimorphism in cuckoo vocalizations (Laymon et al. 1997) without gathering data using banded and known sex cuckoos (Halterman 2002). Current research indicates that vocalizations of cuckoos are variable, with no consistent patterns found with transmitted birds on the San Pedro (Halterman 2002). Unmated birds gave a wide variety of vocalizations, including those formerly attributed to mated birds. Therefore, birds may not be reliably sexed based on vocalizations. Birds are no longer recorded as mated or unmated, male or female, unless clear evidence, like copulation, is observed (Halterman 2003).

Laymon and Halterman (1985) studied the response rate of yellow-billed cuckoos to taped calls during the nesting cycle on the Kern River in California. There is a definite drop in vocalizations both at the start of incubation and several days before hatching. Vocalizations are fairly stable from after hatching (11 days) to fledging (18 days), and an increase in vocalization after fledging (Halterman 2001).

Breeding

Halterman (1991) described breeding behavior for western yellow-billed cuckoos observed along the Sacramento River, California.

> “The formation of mating pairs occurs from late June until mid-July. For several days the newly formed pair searches for a nest site giving frequent kowlping and knocking contact calls. The male will chase other cuckoos at this time but shows little or no agonistic behavior after a nest site has been chosen. The pairs spends two or three days constructing the flimsy twig nest typically located 5 to 40 feet in height in a site with dense canopy cover, and begin incubation as the first of two to four eggs are laid. The female primarily incubates with up to 30% incubation by the male. The birds vocalize infrequently during the 11 to 14 days of incubation, calling only a few times a day during nest exchanges. Both adults care for the young, delivering 20 to 30 food items per day to the nest: katydids (Tetogoniidae), large caterpillars (Lepidoptera), tree frogs (Hyla regilla), and cicadas (Cicadidae). Foraging occurs primarily in the canopy of cottonwood and willow trees. The birds occasionally call while foraging, and usually call when they are approaching the nest with food items. The young leave the nest at 6-8 days; unable to fly crawling from branch to branch [they] may travel up to 50 m on the first day out of the nest, giving soft clucking calls to aid in their location by adults. The adults continue to provide food for 3 to 4 weeks. During this time the young become increasingly mobile and begin to actively follow the foraging adults. The juveniles continue to give their distinctive call, and adults occasionally reply. After this month of parental care the cuckoos will begin their migration through Central America to South America”.

The onset of breeding is apparently correlated with an abundant local food supply or periods of greatest rainfall. Cuckoos may not breed if local food supply is inadequate on breeding grounds following spring migration (Veit and Peterson 1993, Hughes 1999). The breeding cycle is extremely rapid and requires only 17 days from egg-laying to fledging of young. Bursting feather sheaths allow nestlings to become fully feathered within 2 hours (Corman and Magill 2000).

Nesting activities take place between late June and late July, but may begin as early as late May and continue into August, depending on the season. The peak of the breeding season at the South Fork of the Kern River is in the first half of July, although nests have been started as early as June and as late as early August (Laymon 1998). Nest building takes 2-4 days (Bent 1940, Hamilton and Hamilton 1965). However, a transmitted cuckoo on the San Pedro River was observed building a nest in an afternoon, with incubation beginning the next morning (Halterman 2002). One brood of two to three young is raised per season. Cuckoos will occasionally double-brood in western populations if abundant food resources exist, even though the breeding season is 1-3 months shorter than in the east (Hamilton and Hamilton 1965, Hughes 1999).

Nests are oblong, unsymmetrical, loose cup structures of small branches and twigs of willow, mesquite, tamarisk, willow leaves, and caterpillar-eaten cottonwood leaves placed on top of each other, not interwoven. Lining consists of strips of mesquite and cottonwood bark, tamarisk leaves, and rootlets (Groschupf 1987). Cuckoo nests are similar in size and appearance to those of mourning doves, but have more structural integrity (Laymon 1998). Nests are protected from prevailing winds by thick overhanging branches (Hughes 1999).

The average clutch size for cuckoos was 2.95 over the 92 nests on the South Fork of the Kern River. One nest (1.1%) had one egg, 20 (20.2%) had two eggs, 57 (60.6%) had three eggs, 11 (11.7%) had four eggs, and 2 (2.1%) had five eggs. Nests with more than five eggs have been laid by more than one female. The only six-egg clutch was laid by two females, four by the dominant female and two by a secondary female that had her first nest destroyed after her first egg was laid (Laymon 1998). On the Bill Williams River, the average clutch size for 19 nests was 2.3 eggs/nest (Halterman 2001). On the San Pedro River, nest success was 40% (n = 20) in 2004, compared to 86% in 2001, 93% in 2002, and 84.5% in 2003 (Halterman 2005).

Cuckoo offspring experience rapid growth, 17 days from start of incubation to fledgling—among shortest for any species of bird; young gain an average of 4.9 g/day while in the nest (Hughes 1999). Incubation begins with initiation of the first egg laying, known as asynchronous hatching, resulting in eggs and nestlings at different developmental stages in the same nest. Asynchronous hatching permits survival of the oldest nestlings in the event of a food shortage (Gaines and Laymon 1984). The incubation period for yellow-billed cuckoos is 10 to 12 days (Hamilton and Hamilton 1965, Laymon 1998). The young are fed large food items for the 5-8 day nestling period. Most young cuckoos leave the nest on day 6. After fledging, the young are dependent on the adults for at least 2 weeks (Laymon and Halterman 1985).

Yellow-billed cuckoos are loosely territorial, with large overlapping home ranges and little territorial behavior observed (Halterman 2002). Given uniform habitat, they are regularly spaced through the landscape. Along the Sacramento River, in an area of extensive foraging habitat (cottonwoods) and extremely restricted nesting habitat (willows and English walnuts), nests were placed as close as 60 m apart, showing that cuckoos are capable of nesting in close proximity to one another (Laymon 1980). Densities at the South Fork of the Kern River, from 1985 to 1996, averaged 0.85 pairs/40 ha and ranged from a low of 0.15 pairs/40 ha in 1990 to a high of 1.4 pairs/40 ha in 1993 (Laymon 1998). Halterman (1991) found that a multiple regression model, combining patch size, extent of habitat within an 8-km river stretch, presence of point bars, and presence of low woody vegetation, explained 46% of the variance within numbers of cuckoo pairs on the Sacramento River.

On the San Pedro River, home range of two cuckoos fitted with transmitters was estimated at 3 ha and 20 ha. Both cuckoos were observed foraging successfully and feeding fledglings. Based on observations of unbanded cuckoos, the home range of each pair included approximately 2-5 ha of habitat that would be considered poor quality in California (Laymon and Halterman 1989, Halterman 2002). Two radio-tagged nesting birds on the South Fork of the Kern River had minimum home range sizes of 20 ha in 1985, and pairs observed from 1986 to 1989 used home ranges of more than 30 ha (Laymon and Halterman 1990).

The yellow-billed cuckoo has also been observed to breed cooperatively in California, with at least three or four adults tending the nest (Hughes 1999). Preliminary data suggest cuckoos may practice both monogamy and some form of cooperative or sequential polyandry (Halterman 2003, Laymon et al. 1997). Polyandry was recorded for the first time in cuckoos when a female (sex determined by observation of copulation and subsequent monitoring without losing sight of the bird) nested successfully with three males, producing young successfully from three nests (Halterman 2003). Helpers have been observed at more than 30 closely monitored nests on the Kern and Bill Williams rivers, approximately one-third of nests monitored (Laymon et al. 1997, Halterman 2001). In all cases, the helper was a third adult that fed the young multiple times, but these birds were unbanded, so their relationships were unknown.

Typically, yellow-billed cuckoos have one brood per year (Ehrlich et al. 1988). At the South Fork of the Kern River, in years of abundant food resources, two to three broods have been successfully fledged (Laymon et al. 1997). The occurrence of double and triple brooding of yellow-billed cuckoos is significant, indicating that the reproductive potential of the species is much greater than was previously believed. Instead of a pair of cuckoos being able to produce 3-4 young per season, they can actually produce up to 10 young per season, if sufficient food resources are available. At the South Fork of the Kern River, Laymon (1998) found that cuckoos double brooded in less than half of the years, with triple brooding occurring in 1 year of the 12-year study. At the Bill Williams River study site, no evidence of double brooding has been detected (Halterman 2001).

Little is known regarding site fidelity in the yellow-billed cuckoo, but because of apparent movements by cuckoos to take advantage of cicada and tent caterpillar outbreaks in the eastern United States, conventional wisdom is that the species is at least partially nomadic (Robbins et al. 1983). On the San Pedro River, three banded cuckoos were resighted as adults in 2004. In 2002, one bird was seen approximately 1.5 km from its original banding location along the Kern River. On the San Pedro River, observers saw 1 of 2 adults banded in 2001, and 1 of 15 adults banded in 2003. The 2001 cuckoo was seen 25 m from the original banding location, and the 2003 cuckoo was seen approximately 3 km from the original banding location (Halterman 2005).

Fluctuations in yellow-billed cuckoo breeding densities from place to place and year to year have been thought to be attributed to cycles in the abundance of caterpillars, cicadas, and other large insects (Gaines and Laymon 1984). Populations may be highly variable locally (Eaton 1988), depending on food availability, with large localized influxes during times of insect abundances (Hughes 1999). The restriction of mid-summer breeding on the lower Colorado River for the cuckoo is thought to be a response to seasonal peak in large insect abundance, most notably cicadas (Rosenberg et al. 1991.) Presumably, cuckoos are recruited to areas of high caterpillar density (Hamilton and Hamilton 1965). During years of periodic insect outbreaks, cuckoos may be opportunistic, timing nesting to outbreaks rather than to annual emergence of favored insect prey (Nolan and Thompson 1975). Probably more young are raised during years of high caterpillar abundance (Veit and Peterson 1993, Hughes 1999). Cuckoos in Arizona breed at densities considerably lower than apparent food supply would permit, suggesting that spatial distribution and density are dependant on lowest year of food distribution (Hamilton and Hamilton 1965). These thoughts about population fluctuations remain untested, and many questions remain unanswered.

Habitat

Nesting habitat

In Utah, western Texas, New Mexico, Arizona, and southern California, the cuckoo prefers desert riparian woodlands composed of willow (*Salix* spp.), Fremont cottonwood (*Populus fremontii*), and dense mesquite (*Prosopis* spp.) (Hamilton and Hamilton 1965, Gaines 1974) for breeding habitat. Studies from 1987 to 1990 have shown that cuckoos on the Kern River in California nest predominantly in willows, and forage primarily in cottonwoods (Halterman 1991). Radio telemetry of two adults showed that they foraged primarily in cottonwoods, even when willow was the predominant tree present (Laymon and Halterman 1985). Cuckoos on the Kern River preferred habitat patches greater than 15 ha, including at least 3 ha of closed canopy. The average canopy height was 5-30 m, with an understory height of 1-6 m (Laymon and Halterman 1989). Willows, dense low-level or understory foliage, high humidity, and suitable foraging space in excess of 120 m in width and 10 ha in area, characterized most sites from a California statewide survey, including the lower Colorado River and the Bill Williams River (Gaines and Laymon 1984). Cuckoos in Arizona were found along lowland drainages with multistructured native riparian woodlands containing a variable combination of cottonwood, willow, velvet ash, Arizona walnut, mesquite, and tamarisk (Corman 2005).

On the lower Colorado River, the cuckoo must face extremely high midsummer temperatures that would kill unprotected eggs, and, therefore, is likely a nest-site specialist. Mature cottonwoods, with willows forming a subcanopy layer, provide the best shading of any riparian habitat, and provided the optimal habitat on the lower Colorado River. Isolated willows or cottonwoods, mixed with tall mesquites, were also used but to a lesser extent. Standing water in many cottonwood-willow habitats may help lower air temperatures by evaporative cooling (Rosenberg et. al 1991).

Nest sites at the south fork of the Kern River were characterized by higher canopy closure, higher foliage volume, intermediate basal area, and intermediate tree height (Laymon et al. 1997). Sites with less than 40% canopy closure were unsuitable, those with 40-65% closure were marginal to suitable, and those with greater than 65% closure were optimal. Cuckoos seldom used sites that have a foliage volume of less than 20,000

m^3/ha. Optimal nest sites have a foliage volume from 30,000 m^3/ha to 90,000 m^3/ha. Sites with 20,000 m^3/ha to 30,000 m^3/ha or more than 90,000 m^3/ha appear to be suitable. Cuckoos tend to choose nest sites with a mean canopy height of 7-10 m. Sites with a mean canopy height from 4 m to 7 m were chosen less frequently but appear to be suitable, as were sites with a mean canopy height of 10 m to 15 m. Sites with a mean canopy height of less than 4 m were unsuitable. Cuckoos tend to choose nest sites that have a basal area of between 5 m^2/ha and 20 m^2/ha. Sites with basal area 20 m^2/ha to 55 m^2/ha were not used as frequently but were suitable. Sites with basal area less than 5 m^2/ha and over 55 m^2/ha were seldom used by cuckoos and can be considered marginal (Laymon et al. 1997).

In the arid Southwest, breeding populations of yellow-billed cuckoos are restricted to river bottoms, ponds, swampy places, and damp thickets where humidity is relatively high (Hughes 1999). The possibility that nest sites were restricted to river bottoms because of humidity requirements for successful hatching and rearing of young was first suggested in Hamilton and Hamilton (1965). Cuckoos were detected only where surface water was usually present on the Sacramento River (Gaines and Laymon 1984). An analysis of five nest sites on the South Fork of the Kern River and five nest sites on the Sacramento River revealed that temperatures were lower and humidity was higher than surrounding forest patch edges or open areas near forest patch edges (Laymon 2000). Gaines (1974) found that vegetative density, distance to water, and width of the habitat area were important characteristics when surveying for cuckoos.

Foraging habitat

Foraging typically occurred in areas with a greater overall foliage density than in areas where nesting occurs, with an average tree height of 10-15 m (Anderson and Laymon 1989). Of 48 observed foraging attempts, two-thirds were in willows, with the remainder in cottonwood trees. Nearly all attempts were higher than 6 m above ground (Rosenberg et al. 1991). Radio telemetry of two adult cuckoos showed that they foraged primarily in cottonwoods even when willow was the predominant tree present (Laymon and Halterman 1985). Cuckoos with transmitters were observed successfully foraging in mesquite on the San Pedro River (Halterman 2002).

Nest characteristics

The western yellow-billed cuckoo has been associated with cottonwood-willow dominated riparian habitats, with the majority of nests located in willows (*Salix* spp.) and, to a lesser extent, in Fremont cottonwoods (Hamilton and Hamilton 1965; Gaines 1974; Gaines and Laymon 1984; Laymon and Halterman 1985, 1987, 1989; Halterman 1991; Halterman and Laymon 1994; Corman and Magill 2000). Nests on the South Fork of the Kern River were constructed predominately in willows (99%, n = 95). The lone exception was placed in a clump of mistletoe in a cottonwood (Laymon et al. 1997). Nests on the Bill Williams River were constructed in willows (61%, n = 23), tamarisk (35%), and in cottonwood (one nest, 4%) (Halterman 2001). On the Santa Ana River, 22 of 24 nests (92%) were in willows, 1 was in a cottonwood, and 1 was in an alder (Hanna 1937). On the Sacramento River, nests have been found in willows, cottonwoods, boxelders, and rarely in orchards, including prune, English walnut, and almond (Laymon 1998). Several nests on the Sacramento and Santa Ana rivers were draped with wild grape (Hanna 1937). Recent studies in Arizona have found cuckoos nesting successfully in mesquite and hackberry (*Celtis occidentalis*), occasionally some distance away from waterways, but the extent to which they do so in other areas, or even in all years, is unknown (Halterman 2003).

Cuckoos have been found nesting in tamarisk. In 2000 on the Bill Williams River, researchers found three nests in tamarisk, with two cuckoo nests in dense tamarisk stands with no willow or cottonwood present in the vicinity (Halterman 2001). On the lower Colorado River, nests were found occasionally in tamarisk within screwbean mesquite or cottonwood-willow dominated habitats (Rosenberg et al. 1991). Howe (1986) stated that yellow-billed cuckoos nested extensively in tamarisk along the Pecos River in New Mexico.

Cuckoos have also been found nesting in mesquite. On 20 August, 1950, a nest with two eggs was in a small screwbean mesquite 1.8 m above ground on the Bill Williams River (Rosenberg et al. 1991). Cuckoos were found nesting successfully in mesquite and hackberry on the San Pedro River in 2002 (Halterman 2003).

Nests are generally concealed by willow foliage, but are also concealed by other types of vegetation. Several nests at the South Fork of the Kern River (3) and Santa Ana River (1) were concealed in mistletoe. Several nests on the Sacramento River and Santa Ana River were concealed by wild grape (Laymon 1998, Hanna 1937). One nest on the Santa Ana River was in a dead willow and was concealed by poison oak vines (Hanna 1937). Rarely, nests were sufficiently low and herbaceous growth, such as tules, cattails, and goldenrod, was sufficiently tall that it provided nest concealment (Laymon 1998).

Table 1 summarizes yellow-billed cuckoo nest success and nest tree characteristics collected from the Bill Williams River (Halterman 2001), Kern River (Laymon 2000), Lower Colorado River (Rosenberg et al. 1991), and Arizona statewide (Corman 2005).

Table 1. Yellow-billed cuckoo nest success and nest tree characteristics collected from multiple sources

	Number of nests	Mean	SD	Min	Max
Clutch size Bill Williams River	19	2.3	0.5	2	3
Clutch size Kern River	92	2.9		1	5
Number hatched Bill Williams River	18	1.9	0.8	0	3
Nest tree height (m) Bill Williams River	23	9.7	4.3	5	20
Nest tree height (m) Kern River	95	9.4	4.3	2.5	17.8
Nest tree DBH (cm) Bill Williams River	23	19.1	9	7	44
Nest tree DBH (cm) Kern River	85	22.9	13.5	5.8	85.3
Nest height (m) Bill Williams River	23	6.4	3.7	2	17
Nest height (m) Arizona		5.8		1.2	16.8
Nest height (m) Kern River	95	4.8	3	1.3	13
Nest height (m) LCR				4.5	14
Distance from nest tree to forest edge (m) Bill Williams River	23	138.3	167.7	5	500
Distance nest tree to nearest water (m) Bill Williams River	23	287	425.6	0	1000
Distance nest tree to nearest water (m) Kern River	95	310	405.5	0	1500
Nest to branch tip (m) Bill Williams River	23	2	0.9	0.5	4
Nest to tree trunk (m) Bill Williams River	23	1.8	1.9	0	8
Distance to foliage above nest (m) Bill Williams River	23	0.2	0.2	0.1	1
Foliage cover above nest (%) Bill Williams River	23	79.9	14.6	45	100
Foliage cover above nest (%) Kern River	95	93.4	15.1	0	100
Bill Williams River—(Halterman 2001)					
Kern River—(Laymon 2000)					░
Arizona—(Corman 2005)					▒
LCR—(Rosenberg et al. 1991)					▓

Nest site characteristics

At the Bill Williams River, Goodding’s willow (*Salix gooddingii*) made up 48.4% of the tree plots surrounding the nest, tamarisk made up (36.8%), and cottonwood (*Populus fremontii*) made up 12.7% (n = 23) (Halterman 2001). At the South Fork of the Kern River, Goodding's willow (68%), followed by red willow (*Salix laevigata*) (19.1%), and Fremont cottonwood (10.3%) surrounded the nest (Laymon 1998). The dominant shrub species present on the nest site at the South Fork of the Kern River was mule’s fat (*Baccharis salicifolia*) (Laymon 1998). The dominant shrub species data were not collected on the Bill Williams River. Table 2 summarizes yellow-billed cuckoo nest site characteristics from the Bill Williams River (Halterman 2001) and the Kern River (Laymon 2000). These characteristics were measured with the nest tree as the plot center and measurements were taken at 0.1 acre around the nest. Details on how measurements were taken can be found in Halterman 2001.

Table 2. Vegetation survey results taken on 0.1 acre plot centered on nest tree of yellow-billed nest sites from the Bill Williams River and Kern River

Variable	Number of Nests	Mean	SD	Min	Max
Total Canopy cover (%) Bill Williams River	23	79.8	12.1	51	97
Total Canopy cover (%) Kern River	85	74.1	15.6	16.5	98
Canopy Cover Under Nest (%) Bill Williams River	23	93.5	6.8	75	100
Canopy Cover Under Nest (%) Kern River	75	96.8	7.3		
(Taken at 2 locations)					
Canopy Cover 5m from Nest (%) Bill Williams River	23	73.2	17.7	30	97.5
Canopy Cover 5m from Nest (%) Kern River	75	75.1	18.1		
(Taken at 4 locations)					
Canopy Cover 10 m from Nest (%) Bill Williams River	23	70.3	19	35	65
Canopy Cover 10m from Nest (%) Kern River	75	63.8	26.1		
(Taken at 4 locations)					
Total Ground Cover (%) Bill Williams River	23	19	28.6	0	100
Total Bare Ground (%) Bill Williams River	23	69.1	28.6	1	100
Total Bare Ground (%) Kern River	86	23.6	35	0	98
Total Grass Cover (%) Bill Williams River	23	13.4	20.5	0	86
Total Grass Cover (%) Kern River	83	17.3	24	0	100
Total Forb Cover (%) Bill Williams River	23	9.8	23.2	0	95
Total Forb Cover (%) Kern River	85	51.3	33.1	0	100
Total Shrub Cover (%) Bill Williams River	23	33.8	26.2	0	90
Total Shrub Cover (%) Kern River	85	3.5	8.4	0	50
Number of Saplings/ha Bill Williams River	23	0.9	4.1	0	20
Number of Trees/ha Bill Williams River	23	42.5	30	2	132
Number of Gooding's Willow/ha Bill Williams River	23	20.6	17.2	0	57
Number of saltcedar Bill Williams River	23	16.4	30.5	0	132
Number of cottonwood/ha Bill Williams River	23	5.4	9.4	0	32
Foliage Volume (m^3/ha) Bill Williams River	23	78,120	49,499	16,223	222,780
Basal Area (m^2/ha) Bill Williams River	23	18.4	8.9	2.5	42
Mean Quadratic DBH (cm) Bill Williams River	23	18.9	10.5	6	57.2
Mean Height of Trees (m) Bill Williams River	23	8.8	2.4	4.3	14.3
Mean Height of Trees (m) Kern River	83	9.1	2.6	4.4	19.5
Bill Williams River—(Halterman 2001)					
Kern River River—(Laymon 2000)					

Landscape level factors

Patch size is a very important landscape feature for yellow-billed cuckoos. On the Sacramento River from 1987 to 1990, the "extent of habitat in 8 km river stretches" was used as a measure of habitat fragmentation. This was the second-most important variable in determining the presence of pairs ($r^2 = 0.16$, $p < 0.005$), and all cuckoos encountered during this 4-year study ($r^2 = 0.17$, $p < 0.005$) (Halterman 1991). Laymon and Halterman (1989) found that a relationship exists in California between increased size of the habitat patch and the increased proportion of occupied patches. Laymon and Halterman (1989) concluded that sites greater than 80 ha in extent and wider than 600 m were optimal, sites 41-80 ha in extent and wider than 200 m were suitable, sites 20-40 ha in extent and 100-200 m in width were marginal, and sites less than 15 ha in extent and less than 100 m in width were unsuitable.

In California, away from the Colorado River, cuckoos occupied 9.5% of 21 sites that were 20 to 40 ha in extent, 58.8% of 17 sites that were 41 to 80 ha in extent, and 100% of 7 sites greater than 80 ha in extent. The trend toward increased occupancy with increased patch size is significant ($t = 3.63$, $p < 0.001$) (Laymon and Halterman 1989). On the Sacramento River from 1987 to 1990, the extent of patch size was the most important variable in determining occupancy for pairs ($r^2 = 0.25$, $p < 0.005$) and for all cuckoos encountered ($r^2 = 0.27$, $p<0.005$). Patch size, extent of riparian habitat in 8-km river segments, and presence of low woody vegetation explained 47% of the variance in density in nesting yellow-billed cuckoos, using a multiple regression model using pairs of cuckoos as the dependant variable and habitat parameters as the independent variables (Halterman 1991). In California, very few cuckoos were found where suitable vegetation was less than 100 m wide and under 10 ha in surface area (Gaines and Laymon 1984).

In Arizona, several important patch characteristics for yellow-billed cuckoo breeding habitat were identified, including size of patch (40+ ha), minimum width (>200 m, height (>5 m), vegetation type (dense willow/cottonwood riparian), dense canopy cover (>50%), and proximity to surface water (Corman and Magill 2000).

The land adjacent to yellow-billed cuckoo breeding habitat at the South Fork of the Kern River is primarily flood-irrigated pasture and dry range land (Laymon 1998). On the Sacramento River, adjacent habitat varied from dry range land to irrigated farm land and orchards. The distribution of yellow-billed cuckoos at 74 sites along the Sacramento River was not correlated with surrounding land use ($r^2 = 0.03$, $p = 0.59$) (Halterman 1991).

Previous revegetation efforts

On the California side of the Cibola Refuge, an experimental cottonwood-willow revegetation effort began in 1979. The 11-ha site was utilized by foraging cuckoos during the second year. Two nests were found in 1981 and 1982 in planted willows approximately 15 m tall, and thought to represent two or three pairs of cuckoos. In 1983, six cuckoos were detected on the site, with no documented nesting. Cuckoos were absent from the site in 1984 (Anderson and Laymon 1989, Rosenberg et al. 1991).

Threats

Disturbance

During the nest building and egg-laying stages, cuckoos can be very sensitive to human disturbance. Four of 23 nests found on the BWR between 1993 and 2001 were abandoned, with 3 of these likely due to nest-searching efforts (Halterman 2001).

Habitat degradation

Remnant fragments of riparian habitat throughout the West are still endangered by degradation, clearing, and inundation. In Arizona, population declines have been most dramatic along the Gila and lower Colorado rivers, caused primarily by native riparian habitat loss through degradation and fragmentation from lowered water tables, replacement by nonnative trees, grazing practices, and river management (Corman 2005). With small populations fewer than 25 pairs, stochastic events could cause localized extinctions (Laymon and Halterman 1987).

Pesticide use

The effects of pesticides on yellow-billed cuckoos has been little studied. Several studies in Florida showed that yellow-billed cuckoos carried low levels of DDT (0.42 ppm in spring and 1.12 ppm in fall). Eggs taken from two nests in the Sacramento Valley also contained low pesticide levels (0.08 and 0.11 ppm DDE) (Laymon 1980). Yellow-billed cuckoo eggshells collected in the South Fork of the Kern River in 1985 averaged 19% thinner than pre-DDT era eggs, a level of thinning that could cause reproductive failure in some species (Laymon and Halterman 1987). This indicates the possibility that even small pesticide loads in this species can cause significant eggshell thinning. Pesticides, especially larvacides used in mosquito control, could be a major threat when applied on a widespread area (especially aerially). Along the Stanislaus River at Caswell State Park, where cuckoos previously nested, larvacides were regularly applied by air during the spring and summer for many years. This resulted in a loss of insect-eating birds such as warblers, vireos, orioles, flycatchers, and cuckoos (Laymon 1998). The direct effect of pesticide poisoning, especially of cuckoos nesting in or near orchards, can be significant. The young in a nest in a walnut orchard that was sprayed with Zolone for codling moths and aphids could not maintain their balance on tree branches after fledging and repeatedly fell to the ground. This falling behavior has never been observed at dozens of other unsprayed nests (Laymon 1980; Laymon 1998).

Predators

Red-shouldered hawks and northern harriers have been observed preying on nestlings. Cuckoos drive western scrub-jays and loggerhead shrikes away from the nests. On the Sacramento River, there appeared to be an inverse relationship between the presence of cuckoos and scrub jays, indicating a possible aversion by the cuckoos to nesting at sites with western scrub-jays (Laymon 1998). Cooper's hawks are probably the only predator capable of taking adult yellow-billed cuckoos.

LCR MSCP Conservation Measures

YBCU1 – Create 4,050 acres of yellow-billed cuckoo habitat.
Of the 5,940 acres of created cottonwood-willow, at least 4,050 acres will be designed and created to provide habitat for yellow-billed cuckoos. Created habitat will be designed and managed to support cottonwood-willow types I-III that provide breeding habitat for this species. The created cottonwood-willow will also function as migration habitat for birds that migrate along the LCR. A total of 2,700 acres of created habitat will be designed and managed to provide both yellow-billed cuckoo and southwestern willow flycatcher habitat, and 1,350 acres will be designed and managed to specifically provide habitat for the yellow-billed cuckoo.

YBCU2 – Maintain existing important yellow-billed cuckoo habitat areas.
The Applicants, under agreements with cooperating land management agencies, will provide funding to those agencies to maintain a portion of existing yellow-billed cuckoo habitat within the LCR MSCP planning area. Maintaining important existing habitat areas is necessary to ensure the continued existence of the yellow-billed cuckoo in the LCR MSCP planning area, provide for the production of individuals that could disperse to and nest in LCR MSCP created habitats, and reduce the likelihood of future Federal listing of the species.

Management Recommendations

- Create or manage for yellow-billed cuckoo breeding habitat by providing dense foliage, high humidity, extensive habitat, and adequate food sources.

- Create larger areas of suitable habitat restoration. Restoration adjacent to existing large riparian areas is of greater value than creating yet more isolated islands (Halterman 1991). Launer et al. (1990) recommended that restoration efforts in California should be concentrated in areas of sufficient extent to create comparatively large tracts of habitat (a minimum of 100 ha, although smaller patches could be acceptable under certain circumstances).

- Revegetation designs should include patches of willows surrounded by cottonwoods (Anderson and Laymon 1989).

LCR MSCP Research and Monitoring Needs

The western yellow-billed cuckoo has been studied throughout its range; however, information on LCR populations, demographics, habitat use, and threats are not fully understood. System monitoring efforts for yellow-billed cuckoo were initiated in 2006 to determine distribution, demographics, and habitat requirements along the LCR.

Distribution and population trends have not been determined within the LCR MSCP planning area. Monitoring, including banding of individuals, will be conducted to determine potential population trends and distribution.

Yellow-billed cuckoo demographic studies are needed to provide information on site tenacity, site fidelity, nest site selection, reproductive success, and productivity. These data will be used in designing habitat management guidelines for created and existing habitats.

Habitat requirements along the LCR are poorly understood. Studies need to be conducted to determine habitat requirements, including vegetation composition, patch size, micro-habitat characteristics, diet, and prey abundance, for existing and newly created habitats, including habitats used during migration.

Literature Cited

Anderson, B.W., W.C. Hunter, and R. D. Ohmart. Status changes of bird species using revegetated riparian habitats on the Lower Colorado River from 1977 to 1984. California Riparian Systems Conference, September 22-24, 1988, Davis, California.

Anderson, B.W., and S.A. Laymon. 1989. Creating habitat for the yellow-billed cuckoo (*Coccyzus americanus*). USDA Forest Service Gen. Tech. Rep. PSW-110 p. 468-472.

Anderson, B.W., and Ohmart R.D. 1977. Vegetation structure and bird use in the lower Colorado River valley. Importance, Preservation and Management of Riparian Habitat: A Symposium. USDA Forest Service General Technical Report RM-43. Tucson, Arizona.

Banks, R.C. 1988. Geographic variation in the yellow-billed cuckoo. Condor 90:473-477.

Banks, R.C. 1990. Geographic variation in the yellow-billed cuckoo: Corrections and comments. Condor 92:538.

Beal, F.E.L. 1898. Cuckoos and shrikes and their relation to agriculture. U.S. Department of Agriculture Biological Survey Bulletin No. 9.

Bent, A.C. 1940. Life histories of North American cuckoos, goatsuckers, hummingbirds, and their allies. U.S. Natural Museum Bulletin 176.

Corman, T.E., and R.T. Magill. 2000. Western Yellow-billed Cuckoo in Arizona: 1998 and 1999 Survey Report. Nongame and Endangered Wildlife Program Technical Report 150. Arizona Game and Fish Department, Phoenix, Arizona, p. 1-49.

Corman, T.E. 2005. Yellow-billed cuckoo. *In:* Corman, T. E., and Wise-Gervais (editors), The Arizona Breeding Bird Atlas. University of New Mexico Press, Albuquerque.

Ehrlich, P., D. Dobkin, and D. Wheye. 1992. Birds in Jeopardy. Stanford University Press, Stanford, California.

Fleischer, R.C. 2001. Taxonomic and evolutionarily significant unit (ESU) status of western yellow-billed cuckoos (*Coccyzus americanus*). Report to the USGS and USFWS. Molecular Genetics Laboratory, Smithsonian Institution, Washington, DC.

Franzreb, K.E., and S.A. Laymon. 1993. A reassessment of the taxonomic status of the yellow-billed cuckoo. Western Birds 24:17-28.

Fridolfsson, A., and H. Ellegren. 1999. A simple and universal method for molecular sexing of non-ratite birds. Journal of Avian Biology 30:116-121.

Gaines, D. 1974. Review of the status of the yellow-billed cuckoo in California: Sacramento Valley Populations. Condor 76:204-209.

Gaines, D. and Laymon, S.A. 1984. Decline, status and preservation of the yellow-billed cuckoo in California. Western Birds 15:49-80.

Gallagher, T., C.R. Tomlinson, and B. Furtek. 2001. Breeding status of the southwestern willow flycatcher and initial surveys for the Yuma clapper rail and yellow-billed cuckoo at various sites in southern Nevada. Program Activities Report, Nevada Division of Wildlife, Southern Region, Nongame Program.

Griffiths, R., S. Dean, and C. Dijkstra. 1996. Sex identification in birds using two CHD genes. Proceedings of the Royal Society of London 263:1251-1256.

Grinnell, J. 1915. A distributional list of the birds of California. Pacific Coast Avifauna 11.

Grinnell, J., and A.H. Miller. 1944. The distribution of birds in California. Pacific Coast Avifauna 27.

Groschupf, K. 1987. Status of the yellow-billed cuckoo (*Coccyzus americanus occidentalis*) in Arizona and West Texas. Nongame Wildlife Branch, Arizona Game and Fish Department. Contract No. 20181-86-00731.

Halterman, M. 1991. Distribution and habitat use of the yellow-billed cuckoo (*Coccyzus americanus occidentalis*) on the Sacramento River, California, 1987-1990. Masters Thesis. California State University, Chico.

Halterman, M. 2001. Population status of the yellow-billed cuckoo at the Bill Williams River NWR and Alamo Dam, Arizona and southern Nevada: Summer 2000. Prepared for: Bureau of Reclamation, Lower Colorado Regional Office, Boulder City, Nevada.

Halterman, M. 2002. Surveys and life history studies of the yellow-billed cuckoo: Summer 2001. Prepared for: Bureau of Reclamation, Lower Colorado Regional Office, Boulder City, Nevada.

Halterman, M. 2003. Surveys and life history studies of the yellow-billed cuckoo: Summer 2002. Prepared for: Bureau of Reclamation, Lower Colorado Regional Office, Boulder City, Nevada.

Halterman, M. 2004. Surveys and life history studies of the yellow-billed cuckoo: Summer 2003. Prepared for: Bureau of Reclamation, Lower Colorado Regional Office, Boulder City, Nevada.

Halterman, M. 2005. Surveys and life history studies of the yellow-billed cuckoo: Summer 2004. Prepared for: Bureau of Reclamation, Lower Colorado Regional Office, Boulder City, Nevada.

Halterman, M. 2006. Surveys and life history studies of the yellow-billed cuckoo: Summer 2005. Prepared for: Bureau of Reclamation, Lower Colorado Regional Office, Boulder City, Nevada.

Halterman, M.D., D.S. Gilmer, S.A. Laymon, and G.A. Falxa. 2001. Status of the yellow-billed cuckoo in California: 1999-2000. Report to the USGS-BRD Dixon Field Station, Dixon, California.

Halterman, M., M. Johnson, T. Corman, and R. McGill. 2002. Survey methodology and natural history of the yellow-billed cuckoo. Southern Sierra Research Station, Weldon, California.

Halterman, M.D., and S.A. Laymon, 1994. Population status, site tenacity, and habitat requirements of the yellow-billed cuckoo at the Bill Williams River, Arizona: Summer 1993. U.S. Fish and Wildlife Service, Bill Williams National Wildlife Refuge, Parker, Arizona.

Hamilton, W.J. III, and M.E. Hamilton. 1965. Breeding characteristics of the yellow-billed cuckoo in Arizona. Proceedings of the California Academy of Sciences, 4th Series, 32: 405-432.

Hanna, W.C. 1937. California cuckoo in the San Bernardino Valley, California. Condor 39:57-59.

Howe, W.H. 1986. Status of the yellow-billed cuckoo (*Coccyzus americanus*) in New Mexico. Final Report, Contract No. 516.6-75-09, New Mexico Dept. of Game and Fish. Santa Fe, New Mexico. 16 p. + appendices.

Hughes, J.M. 1999. Yellow-billed cuckoo (*Coccyzus americanus*). *In:* A. Poole and F. Gill (editors). The Birds of North America, No. 418. The Birds of North America, Inc., Philadelphia, Pennsylvania.

Johnson, M.J., Holmes, J.A, and Weber, R. 2005. Yellow-billed cuckoo distribution and abundance, habitat use, and breeding ecology along the lower Colorado River (Yuma, Arizona-United States/Mexico Border), Cibola NWR, Imperial NWR, Picacho State Recreation Area, California, Mittry Lake WMA, Colorado/Gila River Confluence, Gila River, and Quigley Pond WMA, 2005. Northern Arizona University.

Kahn, N.W., J. St. John, and T.W. Quinn. 1998. Chromosome-specific intron size differences in the avian CHD gene provide an efficient method for sex identification in birds. Auk 115:1074-1078.

Koronkiewicz, T.J., M.A. McLeod, B.T. Brown, and S.W. Carothers. 2004. Southwestern willow flycatcher surveys, demography, and ecology along the lower Colorado River and tributaries, 2003. Annual reports to Bureau of Reclamation, Boulder City, Nevada, by SWCA Environmental Consultants, Flagstaff, Arizona.

Koronkiewicz, T.J., M.A. McLeod, B.T. Brown, and S.W. Carothers. 2005. Southwestern willow flycatcher surveys, demography, and ecology along the lower Colorado River and tributaries, 2004. Annual reports to Bureau of Reclamation, Boulder City, Nevada, by SWCA Environmental Consultants, Flagstaff, Arizona.

Koronkiewicz, T.J., M.A. McLeod, B.T. Brown, and S.W. Carothers. 2006. Southwestern willow flycatcher surveys, demography, and ecology along the lower Colorado River and tributaries, 2005. Annual reports to Bureau of Reclamation, Boulder City, Nevada, by SWCA Environmental Consultants, Flagstaff, Arizona.

Latta, M.J., C.J. Beardmore, and T.E. Corman. 1999. Arizona partners in flight bird conservation plan (Technical Report 142). Nongame and Endangered Wildlife Program. Arizona Game and Fish Department. p. 193-197.

Launer, A.E., D.D. Murphy, S.A. Laymon, and M.D. Halterman. 1990. 1990 distribution and habitat requirements of the yellow-billed cuckoo in California. Center for Conservation Biology, Department of Biological Sciences, Stanford University, Stanford, California. Prepared for Kern River Research Center. P.O Box 990, Weldon, CA 93283.

Laymon, S.A., and M.D. Halterman, 1985. Yellow-billed cuckoos in the Kern River valley: 1985 population, habitat use, and management recommendations. Prepared for the Nature Conservancy, Kern River preserve. California Department of Fish and Game, Nongame Bird and Mammal Section, Sacramento, California.

Laymon, S.A., and M.D. Halterman. 1987. Can the western subspecies of the yellow-billed cuckoo be saved from extinction? Western Birds 18:19-25.

Laymon, S.A., and M.D. Halterman. 1989. A proposed habitat management plan for yellow-billed cuckoos in California. USDA Forest Service Gen. Tech. Rep. PSW-110. p. 272-277.

Laymon, S.A., P.L. Williams, and M.D. Halterman. 1997. Breeding status of the yellow-billed cuckoo in the South Fork of the Kern River Valley, Kern County, California: Summary Report 1985-1996. Prepared for USDA Forest Service, Sequoia National Forest, Cannell Meadow Ranger District. Challenge Cost-share Grant #92-5-13.

Laymon, S.A. 2000. Supporting information for the listing of the western yellow-billed cuckoo. Prepared for: U.S. Fish and Wildlife Service, Sacramento Fish and Wildlife Office, Sacramento, California.

Manolis, T., B. Webb, R. Spotts, S. Evans, B. Andrews, C. Brown, R. Schmidt, A. Tura, and M.J. Palmer. 1986. Petition to list the western yellow-billed cuckoo as endangered in a significant portion of its range. Letter to Frank H. Dunkle, Director, U.S. Fish and Wildlife Service.

McKernan, R.L, and G. Braden. 2002. Status, distribution, and habitat affinities of the southwestern willow flycatcher along the lower Colorado River, Year 6—2001. Submitted to Bureau of Reclamation, Lower Colorado River Region, Boulder City, Nevada.

Potter, E.F. 1980. Notes on nesting yellow-billed cuckoos. Journal of Field Ornithology 51:17-29.

Pruett, C.L., D.D. Gibson, and K. Winker. 2001. Molecular “Cuckoo Clock” suggests listing of the western yellow-billed cuckoos may be warranted. The Wilson Bulletin:228-231.

Pyle, P., N.G. Howell, R.P. Yunick, and D.F. DeSante. 1997. Identification guide to North American passerines: Part 1. Slate Creek Press, Bolinas, California. 730 p.

Ridgeway, R. 1887. A manual of North American birds. Lippincott, Philadelphia.

Rosenberg, K.V., R.D. Ohmart, W.C. Hunter, and B.W. Anderson. 1991. Birds of the Colorado River Valley. University of Arizona Press. Tucson, Arizona.

Suckling, K., and Greenwald, D.N. 1998. Petition to list the yellow-billed cuckoo as a Federally endangered species. Southwest Center for Biological Diversity. Tucson, Arizona.

Stephens, F. 1903. Bird notes from eastern California to western Arizona. Condor 5:75-78.

U.S. Fish and Wildlife Service. 2000. Endangered and threatened wildlife and plants: Notice of 90-day finding for a petition to list the yellow-billed cuckoo as endangered and commencement of a status review. Federal Register Vol. 65, No. 33, Feb. 17, 2000.

U.S. Fish and Wildlife Service. 2001. 12-month finding for a petition to list the yellow-billed cuckoo (*Coccyzus americanus*) in the western continental United States. Federal Register Vol. 66, No. 143, 38611-38626.

Veit, R., and W. Petersen. 1993. Birds of Massachusetts. Massachusetts Audubon Society. Lincoln, Massachusetts.

ELF OWL
(*Micrathene whitneyi*)

Legal Status

The elf owl is not a Federally listed species. The state of California listed the elf owl as endangered in 1980 (CDFG 1980). The elf owl is not listed in either Arizona or Nevada.

Distribution

Historical Range

Elf owl fossil history in southern Arizona is known from 6,080 ± 250 years ago (Mead et al. 1984) and 11,100 ± 300 years ago (Van Devender et al. 1991). Fossils records from 11,000-27,000 years ago were also discovered in Nuevo León, Mexico (Steadman et al. 1994). Henry and Gehlbach (1999) suggest that the elf owl occupied evergreen woodlands and riparian forests in the present Sonoran Desert region before saguaro (*Carnegiea gigantea*) cactus appeared 8,000 years ago.

Current Range

Elf owls are known to breed in three distinct areas of the southwestern United States (Figure 1):

1. The lower Colorado River, from southern Nevada, eastern California, and western Arizona, west to the Rio Grande River in New Mexico.
2. The Big Bend region of Texas, east to Edwards Plateau.
3. Dimmit County, Texas, southward, through the Rio Grande River, to Nuevo León, Mexico.

In Mexico, there are three distinct year-round and wintering populations of elf owls (Figure 1) including:

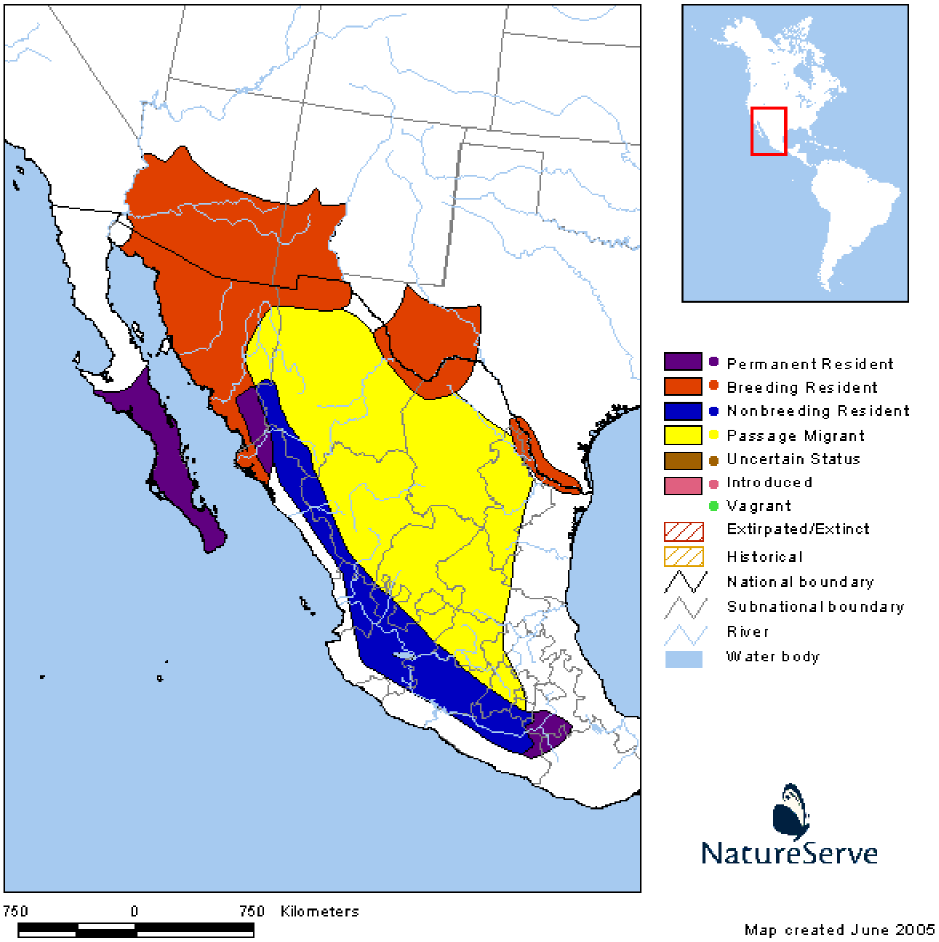
4. The southern portion of Baja California, which supports a year-round population.
5. Socorro and other Revilla Gigedo Islands, which support a year-round population.
6. A wintering population located from southwestern Puebla to northern Oaxaca.

Populations within LCR MSCP Project Boundary

In the past 100 years, elf owl presence in the LCR MSCP area has been sparse. In the early 1900s, as the United States was beginning to develop the LCR, naturalists traveled the river documenting hundreds of species. Most of these records are just documentation with no scientific surveys performed. Scientific surveys for elf owls were first conducted in the 1970s and 1980s.

Brown reported observing elf owls near Yuma, Arizona (1903), and in Duncan Flats and Senator Mine in California opposite Yuma (1904). Stephens (1903) observed an elf owl near Needles, California, on the Arizona side of the river. Kimball (1922) reported elf owls utilizing cottonwood (*Populus* spp.) trees in Bard, California. Elf owls were also reported in the Kofa Mountains east of Yuma (Walker 1943). Miller (1946) reported a pair of elf owls in a cottonwood tree at Joshua Tree National Park, 70 miles west of the LCR. Cardiff (1978, 1979) surveyed the LCR and the western bank (for the state of California) for elf owls, finding 10 pairs in 1978 and 5 pairs in 1979. Halterman et al. (1987) also surveyed the LCR and western bank (for the state of California), finding 15-18 pairs of elf owls. Halterman et al.'s (1987) higher numbers may be attributed to the fact that they surveyed almost twice as many sites as Cardiff (1978, 1979). McKernan and Branden (2002) reported observing elf owls in multiple locations along the LCR, from Lake Havasu (Lake Havasu City, Arizona) south to Taylor Lake (20 miles north of Yuma, Arizona) during southwestern willow flycatcher surveys. Data from the Arizona Breeding Bird Atlas (Wise-Gervais 2005) describe elf owls at the Bill Williams River delta. Figure 2 summarizes elf owl observations along the LCR.

Figure 1. Elf Owl Distribution.



Map created June 2005

Table 1. Elf Owl Observations along the lower Colorado River

Area	Elf Owls Observed	Citation
Needles, CA	3+	Stephens 1903
Needles, CA-Yuma, AZ	20, 10, 30-36	Cardiff 1978, 1979, Halterman et al. 1987
Joshua Tree NP, Riverside County, CA	2	Miller 1946
Lake Havasu south to Taylor Lake	unknown	McKernan and Braden 2002
Bill Williams River Delta, AZ	unknown	Wise-Gervais 2005
Kofa Mountains, Yuma, AZ	5	Walker 1943
Yuma, AZ	5, 5	Brown 1903, 1904
Bard, CA	2	Kimball 1922

Life History

General Description

Elf owls are the smallest owl in the world, with adults measuring 12.4-14.2 cm in length and weighing 35-55 g. Its back is grayish brown with buff mottling; the belly is grayish white with cinnamon vertical streaks. A prominent white eyebrow stripe encircles cinnamon facial disks, there are no ear tufts, and the iris is yellow. The wings have two white scapular stripes. Elf owls have a short tail with buffy bands. The sexes are alike in plumage, with the female unnoticeably larger. The plumages remain the same throughout the year. Four months after hatching, juvenile plumage is mottled grayish brown and resembles the adult. Elf owls cannot be confused with other owls as other owls are noticeably larger and differ greatly in plumage colorization. Elf owls are nocturnal with higher rates of activity from sunset plus 4 hours and from 4 hours until sunrise.

Vocalizations

The developing nestling makes a soft peep or squeak, and a twitter followed by a rasp, approximately 48 times per minute, with volume proportional to their hunger (Gilman 1909, Ligon 1968). Hardy et al. (1999) reported hearing the rasps up to 100 m away. During feeding, nestlings give high-pitched trills (Ligon 1968). Nestlings develop amateurish vocalizations that progress until their first year as adults (Henry and Gehlbach 1999).

Adult males perform a chatter song comprising 5-7 notes, delivered at the rate of 5-6 notes/second, with a faint beginning note rising with emphasis at the end. The song resembles a dog yipping. Elf owls perform this chatter song as a nest site proclamation/defense and advertisement by the male to the female. The chatter song varies in frequency and becomes more insistent when humans or conspecifics are near. Another version of the chatter song is 8-9 notes/second, lasting more than 1 minute, increasing and decreasing with volume. Males perform this song for the female as intense nest cavity advertisement. In flight, the males advertise to the females with a *CHUR-ur-ur-ur* increasing in rate and volume from one series to another (Ligon 1968).

Adult females utter a shrill *sheeee* during copulation (Ligon 1968) and a cricket-like *rrrrrrr* when fed by the male, similar to the nestlings trill (Miller 1946).

Both sexes vocalize a soft, whistle-like, slurred single call note *peeu*. The pairs perform this call between each other during the feeding of the nestlings as contact calls. Females often initiate this call for pair and family contact. The adults bark a single sharp *cheur* (Ligon 1968) repeated rapidly as an alarm call when danger is present (Boal et al. 1997). Elf owls readily respond to imitated calls from humans, as well as vocalizations from conspecifics and predators (Ligon 1968, Goad and Mannan 1987, Boal and Bibles 2001).

Elf owls sing regularly in the spring during pair formation and through the summer when nestlings fledge. Song frequency is directly proportional with the full moon cycles and clear skies (Ligon 1968, Goad and Mannan 1987). Vocalizations typically begin at dusk, decreasing into the night and increasing again as dawn approaches. Song can continue uninterrupted for an hour or more (Henry and Gehlbach 1999). Elf owls elicit these vocalizations from perches, nests, and in flight (Ligon 1968).

Habitat

In most of its breeding range, the elf owl is associated with mature saguaro cactus (*Carnegiea gigantae*; Stephens 1903, Campbell 1934). Along the LCR, elf owls are associated with mesquite (*Prosopis* spp.) woodlands and cottonwood-willow (*Salix* spp.) riparian areas (Gilman 1909, Kimball 1922, Miller 1946, Halterman et al. 1987). In the non-breeding season, elf owls utilize tropical deciduous forests with columnar cacti (*Stenocereus* spp. and *Neobuxbaumia* spp.) and arid grassy savanna with columnar cacti (*Lemaireocereus* spp., Henry and Gehlbach 1999).

Diet

The major food items are arthropods from 22 families, consisting principally of insects, including moths, beetles, and crickets (Henry and Gehlbach 1999). The prey type changes to primarily scarab beetles as the summer rains bring large population of these beetles (Ligon 1968). There have been a few reports of elf owls eating snakes, lizards, and mice (Ligon 1968). In the nesting season, adults often cache prey for later consumption by nestlings (Ligon 1968, Henry and Gehlbach 1999).

Elf owls capture their prey in flight, on the ground, on foliage, on flowers, around outdoor lights, and at hummingbird feeders. Elf owls capture their prey with their beak or feet via a sit and wait strategy from strategic perches. Occasionally, they run after the prey on the ground. Elf owls probe for insects on flowers (Walker 1943, Marshall 1957, Ligon 1968). Ligon (1968) reported elf owls removing scorpion stingers and dangerous terminal abdominal ends in order to avoid poisonous parts.

Behavior

Adult flight consists of rapid, uniform wing beats typically in a straight line. Flights between perches resemble an arc, typical of other cavity nesters (Ligon 1968). Elf owls are occasionally seen gliding or hovering (Walker 1943). Fledglings fly well out of the nest (Ligon 1968). Henry and Gehlbach (1999) report adults occasionally walking, hopping, and running after prey.

Little is know about elf owl self-maintenance, whereas preening and allopreening are widely reported in most other owls (Forsman and Wright 1979, pers. obs.).

To avoid detection, the elf owl stands erect and compressed, with one wing partially covering its face, and its side toward the intruder, following the intruder’s movement. When threatened, the elf owl erects its breast contour feathers, lowers one or both wings slightly, sways its body, and claps its bill (Ligon 1968). Nesting females have feigned death when approached by humans (Brown 1903, Ligon 1968).

Elf owl home range is from 0.2 to 0.4 ha, with a home range overlap of up to 20% with other elf owls (Ligon 1968). Males are polyterritorial; they defend more than one cavity but not the space between the cavities (Ligon 1968, Goad and Mannan 1987). Both sexes defend nests vocally, and against conspecifics (Ligon 1968, Boal et al. 1997). Ligon (1968) suggests that male elf owls defend against the number of other elf owls in adjacent territories but not individuals. This suggests that elf owls do not have an intraspecies hierarchy and compete only with the number of individual elf owls. Ligon observed territorial behavior caused by an individual’s own recorded and broadcasted vocalizations.

Elf owls display many degrees of sociality. They flock during migration, small fledging elf owls group together, and adults cooperatively mob predators (Ligon 1968).

Migration

In the LCR MSCP area, elf owls return to breeding grounds in March and leave in September (Phillips 1942, Phillips et al. 1964). Elf owls spend the non-breeding season in Mexico as far south as Oaxaca. Ligon (1968) suggests that elf owl migrate because arthropod food resources are more active farther south during the winter.

Threats

Great horned owls and Cooper’s hawks predate on adults and fledglings (Ligon 1968). In defense from predators, elf owls cooperatively mob, incessantly vocalize, and physically attack (Boal et al. 1997). Ligon (1968) reports fly larvae parasitizing elf owls.

Breeding

The elf owl is a secondary cavity nester, relying on cavities excavated by other birds. Throughout the LCR MSCP area, elf owls utilize cavities in cottonwood, willow, saguaro, and *Tamarix* spp. trees, formerly occupied by Gila woodpeckers (*Melanerpes uropygialis*), gilded flickers (*Colaptes chrysoides*), and ladder-backed woodpeckers (*Picoides scalaris*; Halterman et al. 1987).

Males arrive ahead of females on the breeding grounds in mid-March. By May, males select and advertise more than one potential cavity for the yet-unmated females to choose (Ligon 1968). Cavity orientation in south central Arizona is random (Goad and Mannan 1987); however, in southwestern Arizona, at lower elevations, cavities are oriented north (Hardy and Morrison 2001). This difference in orientation may be due to the higher temperatures in southwestern Arizona. Nest site competition in southern Arizona does not present a problem for elf owls (Ligon 1968, Bibles 1992 *in* Henry and Gehlbach 1999).

Typically, pair formation is established when the female accepts food from the male at a cavity of her choice, typically in early May. Pair bonds last for approximately 3 months (Ligon 1968). Copulation occurs after cavity selection. Typically, copulation occurs multiple times per night over several nights. The female gives a distinctive *shee* vocalization during copulation (Ligon 1968).

The female may choose the nest cavity weeks before egg laying occurs. This is presumably to retain the cavity from other cavity nesters. From the start of cavity selection, the male feeds the female increasingly through the fledging of young. Nests are bare wood bottoms of the cavities. Removal of primary nests and debris is common (Ligon 1968, Henry and Gehlbach 1999).

Nest cavity height in cottonwood-willow/mesquite habitat is unknown. In southern Arizona saguaros, nest height averages 6 m (Goad and Mannan 1987). In the mountain canyon of southeastern Arizona, nest height averaged 10 m in sycamore trees (Ligon 1968). Nest cavity microclimate is more stable than the outside ambient temperature (Ligon 1968, Henry and Gehlbach 1999).

Elf owls have one brood and replace lost clutches (Ligon 1968, Henry and Gehlbach 1999). Elf owls lay eggs in late April to late June (Brown 1903, Ligon 1968, Henry and Gehlbach 1999). This discrepancy in timing may be due to local and climatic temperature differences. Three eggs are typically laid in primary nests over 6 days, with 1-2 eggs laid in replacement nests (Ligon 1968, Henry and Gehlbach 1999). The female solely incubates the eggs for 24 days (Ligon 1968, Henry and Gehlbach 1999). The female remains on the nest for all but brief periods at dusk and dawn. The male feeds the female at the cavity entrance (Ligon 1968).

Growth of the young is rapid. The hatched young stay in the nest for 28-33 days, gaining 40 g of weight in that short time. The young call from the nest as the male brings food to the cavity entrance and transfers it to the female who, in turn, feeds the young. Ligon (1968) observed intensive and seemingly unending feeding, upwards of once a minute, 569 times a week. The adults do not remove nest debris and the nests can quickly become foul smelling (Ligon 1968).

Young depart the nest 28-33 days after hatching, around dusk. Adults may call young from outside the nest to encourage flight. Withholding food from the young is a common practice of enticing flight. The fledging flight is cautious but good. Fledglings can capture prey immediately but they also beg for food from adults. How long fledglings depend on the adults is unknown, as is their behavior during the immature stage (Ligon 1968).

Demography and Populations

Females breed in the first summer succeeding hatching. The timing of initial male breeding is unknown. Annual reproductive success of nests ranges from 50 to 90%. Fledgling success is at 90%. These rates are higher than reproductive and fledgling success rates for other owls and passerines (Henry and Gehlbach 1999). The oldest age recorded for an elf owl was 4 years 11 months in the wild (Klimkiewicz and Futcher 1989), and 14 years in captivity (Henry and Gehlbach 1999).

Breeding density varies between habitat types, from 2.2 to 5.0 pairs/km^2. The lower densities occur at the edges of the elf owl habitat and the higher rates are associated with the subtropical, riparian woodlands (Henry and Gehlbach 1999). Densities along the LCR are unknown.

Populations may be regulated by rainfall. Henry and Gehlbach (1999) suggest that higher rainfall amounts and higher temperatures increase the prey base (arthropods), increasing the number of breeding elf owls and increasing productivity. A 3-year nest box study yielded 23-34% nest box use, with 51-76% productivity, when rainfall averaged 10-18 cm. In a drier year (6 cm rainfall), nest box use declined 6%, but productivity decreased 52% (McKinney 1996).

Survey Protocol

Currently, a standardized survey protocol does not exist. Hardy and Morrison (2000) used a call playback method to survey elf owls. They established transects and broadcast calls at stations every 0.8 km. Calls were broadcast at 100-110 db at 1 m. Surveys were performed between mid-March through late May. Surveys began 30 minutes after sunset and were completed within 4 hours. At each station, there was a 2-minute settling period, followed by a 2-minute count period when all detections were noted. They then played the species song for 30 seconds, rotating the megaphone 360 degrees, followed by a 2-minute silent count period. Hardy and Morrison (2000) broadcast great horned owls calls along with the elf owl calls. Boal and Bibles (2001) showed that elf owls respond to great horned owl calls during the nestling stage of the breeding season.

Conservation and Management

The decline of elf owl habitat is not contested; however, it is difficult to know exactly how the past 100 years of LCR management has affected the elf owl. As stated above, there have been sporadic confirmed populations of elf owls along the LCR; however, no system-wide elf owl surveys were conducted until Cardiff (1978). Halterman et al. (1987) located more elf owls than Cardiff (1978); however, Halterman et al. (1987) surveyed twice as many sites.

In 1986, The Santa Cruz Predatory Bird Research Group attempted to reintroduce elf owls outside of Needles, California. Attempts to attach transmitters and relocate the owls in 1988 were unsuccessful (Henry and Gehlbach 1999). In Texas, Gamel (1997) had success with radio transmitters in relocating elf owls.

The effects of pesticides, hunting, shooting, and collisions with human-made structures on elf owls are not known. Researchers and birders may have a negative affect on elf owls by imitating vocalizations, shining lights on nests, and beating on trees to determine if elf owls are in cavities.

Nest Box Installation

McKinney (1996) in Texas added nest boxes in elf owl habitat to determine if the owls would utilize the artificial nest cavities. Boxes were modeled after ladder-backed woodpecker cavity dimensions. The boxes were made of rough pine, 30.5 cm tall, 13.9 cm wide, and 13.9 cm deep, with an entrance hole 6.3 cm in diameter, 22.8 cm above the floor. Within 1 year, the elf owls were utilizing 30% of the nest boxes. McKinney (1996) found that an average of 25% of elf owl nest boxes were used over a 4-year study.

LCR MSCP Conservation Measures

MRM3 – Conduct research to determine and assess the effects of the nest site competition with European starlings on reproduction of covered species.
Research will be undertaken to determine whether nest site competition with European starlings is a substantial factor limiting the reproductive success of the elf owl, gilded flicker, and Gila woodpecker. If so, experimental programs may be implemented to determine the effectiveness and practicality of controlling starlings.

ELOW1 – Create 1,784 acres of elf owl habitat.
Of the 7,260 acres of created cottonwood-willow and honey mesquite land cover, at least 1,784 acres will be designed and created to provide elf owl habitat. Patches of created habitat will be designed and managed to support cottonwood-willow types I and II and honey mesquite type III that provide habitat for this species. The created habitat will be established in patches as large as possible. At a minimum, however, isolated patches of honey mesquite type III will be created in patches of at least 50 acres, and of the 5,940 acres of LCR MSCP-created cottonwood-willow, 1,702 acres will be created in patches of at least 50 acres, 2,348 acres will be created in patches of at least 25 acres, and 1,890 acres will be created in patches of at least 10 acres. In addition to the spatial replacement of affected habitat, the quality of created habitat will be substantially greater than affected habitats; thus, created habitat will approximate the condition of native habitat of the species that was historically present along the LCR.

ELOW2 – Install elf owl nest boxes.
Until vegetation has matured sufficiently to attract woodpeckers that are needed to create nesting cavities for the elf owl, structural characteristics of nesting habitat (i.e., snags) will be artificially established. Installation of 2-5 nest boxes on poles or sufficiently tall trees per 250 acres of created habitat will be conducted to replicate the average breeding density of established populations in the southwestern United States (Henry and Gehlbach 1999; LCR MSCP 2004)

LCR MSCP Research and Monitoring Needs

Little is known about elf owl populations, distribution, demographics, and habitat requirements within cottonwood-willow habitats along the LCR. Standardized survey protocol has not been developed. Once a survey protocol has been established, distribution and population occurrence can be determined.

Elf owl habitat requirements within riparian habitat needs to be defined. Vegetation composition, structure, density, minimum patch size, and habitat fragmentation need to be determined. Demographic data needs to be collected, including fledgling dispersal, prey abundance, cavity selection, and competition with other cavity nesters (European starlings). Colonization rates within habitat creation sites should be compared with other cavity nesting species.

Nest boxes may be used to artificially create cavities in created habitats. Nest box use should be studied to determine if the boxes adequately substitute for natural nesting cavities. Alternate methods for producing natural cavities should be investigated.

Literature Cited

Bibles, B.D. 1992. Is there competition between exotic and native cavity-nesting birds in the Sonoran Desert: An experiment. M.S. Thesis, University of Arizona, Tucson.

Boal, C.W., B.D. Bibles, and R.W. Mannan. 1997. Nest defense and mobbing behavior of elf owls. Journal of Raptor Research 31:286-287.

Boal, C.W., and B.D. Bibles. 2001. Responsiveness of elf owls to conspecific and great horned owl calls. Journal of Field Ornithology 72:66-71.

Brown, H. 1903. Arizona bird notes. The Auk 20:43-50.

Brown, H. 1904. The elf owl in California. The Condor 6:45-47.

California Department of Fish and Game. 1980. Endangered and threatened animals of California. Habitat Conservation Planning Branch, California Department of Fish and Game, Sacramento.

Campbell, B. 1934. Bird notes from southern Arizona. The Condor 36:201-203.

Cardiff, S. W. 1978. Status of the Elf Owl in California. Progress Report to Nongame and Mammal Section, Wildlife Management Division, California Department of Fish and Game.

Cardiff, S.W. 1979. Status of the elf owl in California. Final Report to Nongame and Mammal Section, Wildlife Management Division, California Department of Fish and Game.

Forsman, E.D., and H.M. Wright. 1979. Allopreening in owls: What are its functions? The Auk 96:525-531.

Gamel, C.M. 1997. Habitat selection, population density, and home range of the elf owl, *Micrathene whitneyi*, at Santa Ana National Wildlife Refuge, Texas. M. S. Thesis, University of Texas-Pan American, Edinburg, TX.

Gilman, M.R. 1909. Some owls along the Gila River in Arizona. The Condor 11:45-150.

Goad, M.S., and R.W. Mannan. 1987. Nest-site selection by elf owls in Saguaro National Monument, Arizona. The Condor 89:659-662.

Halterman, M.D., S.A. Laymon, and M.J. Whitfield. 1987. Population assessment of the elf owl in California. Final Report to Nongame and Mammal Section, Wildlife Management Division, California Department of Fish and Game.

Hardy, P.C., M.L. Morrison, and R.X. Barry. 1999. Abundance and habitat association of elf owls and western screech owls in the Sonoran Desert. The Southwestern Naturalist 44:311-323.

Hardy, P.C., and M.L. Morrison. 2000. Factors affecting the detection of elf owls and western screech owls. Wildlife Society Bulletin 28:333-342.

Hardy, P.C., and M.L. Morrison. 2001. Nest site selection by elf owls in the Sonoran Desert. The Wilson Bulletin 113:23-32.

Henry, S.G., and F.R. Gehlbach. 1999. Elf owl (*Micrathene whitneyi*). *In:* A. Poole and F. Gill (editors), The Birds of North America, No. 413. The Birds of North America, Inc. Philadelphia, Pennsylvania.

Kimball, H.H. Bird records from California, Arizona, and Guadalupe Islands. The Condor 24:96-97.

Klimkiewicz, M.K., and A.G. Futcher. 1989. Longevity records of North American birds. Supplement 1. Journal of Field Ornithology 60:469-494.

Ligon, J.D. 1968. The biology of the elf owl, *Micrathene whitneyi*. University of Michigan Museum of Zoology. Miscellaneous Publication 136.

Lower Colorado River Multi-Species Conservation Program. 2004. Lower Colorado River Multi-Species Conservation Program, Volume II: Habitat Conservation Plan. Final. December 17. (J&S 00450.00.) Sacramento, California.

Marshall, J.T., Jr. 1957. Birds of the pine-oak woodland in southern Arizona and adjacent Mexico. Pacific Coast Avifauna No. 32.

McKernan, R.L., and G. Branden. 2002. Status, distribution, and habitat affinities of the southwestern willow flycatcher along the lower Colorado River year 6 – 2001. Submitted to Bureau of Reclamation, Lower Colorado River Region, Boulder City, Nevada, and USFWS, Carlsbad Field Office, California, and USFWS Reno Office, Nevada.

McKinney, B.R. 1996. The use of artificial nest sites by elf owls in western Texas. Pages 38-39 *In:* Wildlife research highlights 1996 Volume 1. Texas Parks and Wildlife Department, Austin.

Mead J.L., E.L. Roth, T.R. Van Devender, and D.W. Steadman. 1984. The late Wisconsinan vertebrate fauna from Deadman Cave, southern Arizona. Transactions of The San Diego Society of Natural History 20:247-276.

Miller, L. 1946. The elf owl moves west. The Condor 48:284-285.

Phillips, A.R. 1942. Notes of the migration of the elf owl and flamulated screech owls. The Wilson Bulletin. 54:132-137.

Phillips, A.R., J. Marshall, and G. Monson. 1964. The birds of Arizona. University of Arizona Press, Tucson.

Steadman, D.W., J. Arroyo-Cabrales, E. Johnson, and A.F. Guzman. 1994. New information on the late Pleistocene birds from San Josecito Cave, Nuevo León, Mexico. The Condor 7:577-589.

Stephens, F. 1903. Bird notes from eastern California and western Arizona (concluded). The Condor 5:100-105.

Van Devender, T.R., J.I. Mead, and A.M. Rea. 1991. Late Quaternary plants and vertebrates from Picacho Peak, Arizona. The Southwestern Naturalist 36:302-314.

Walker, L.W. 1943. Nocturnal observations of elf owls. The Condor 45:165-167.

Wise-Gervais, C. 2005. Elf Owl. Pages 220-221 *In:* Corman, T. E., and C. Wise-Gervais (editors), Arizona Breeding Bird Atlas. University of New Mexico Press, Albuquerque.

GILDED FLICKER
(*Colaptes Chrysoides*)

Introduction

A review of the literature indicates that very little research has been conducted on the gilded flicker (*Colaptes chrysoides*). Most of the available research has been conducted on the northern flicker (*Colaptes auratus*) and its subspecies, the red-shafted flicker (C. *a*. *cafer*) and the yellow-shafted flicker (*C*. *a*. *auratus*). The American Ornithologists Union has considered the gilded flicker a subspecies of, or the same species as, the northern flicker several times in the past. Therefore, past research on the northern flicker in the Southwest may have actually been conducted on the gilded flicker; however, this cannot be substantiated. In this document, the term flicker will refer to the *Colaptes* genus.

Legal Status

The gilded flicker is not a Federally listed species. The state of California listed the gilded flicker as endangered in 1988 (CDFG 1988). The gilded flicker is not listed in either Arizona or Nevada.

Distribution

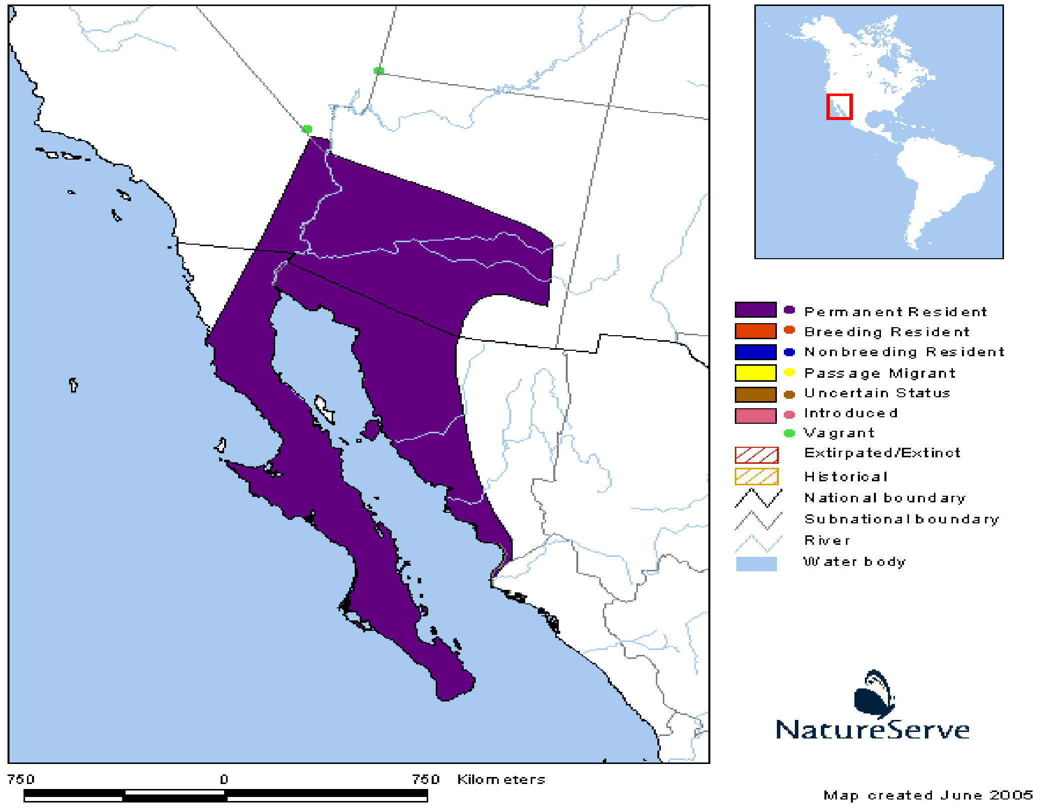
Historical Range

The historical range of the gilded flicker has not been adequately described. Taxonomic differences within the genus *Colaptes* have been confounded over the past 100 years. The fossil record indicates that the gilded flicker may have been present in California at least 12,000 years ago (Miller and Demay 1942). These records are within the gilded flicker's range; however, the records are for the genus *Colaptes* and not necessarily for *C*. *chrysoides*.

Current Range

Gilded flickers breed mostly where saguaros (*Carnegiea gigantea*) and Joshua trees (*Yucca brevifolia*) exist along the lower Colorado River in southern Nevada, western Arizona, and eastern California, south to the tip of Baja California, Mexico, and west through the Sonoran Desert (Figure 1.). Typically, gilded flickers are absent in urban areas within their range.

Figure 1. Gilded Flicker Distribution



Populations within LCR MSCP Project Boundary

At Fort Mohave, Arizona, Cooper (1870 *in* Grinnell 1914) found two pairs of gilded flickers in cottonwood (*Populus* spp.) trees. Grinnell (1914) found two pairs in dead cottonwood stumps just north of Laguna Dam in the Potholes region. Grinnell (1914) noted many nest cavities in the saguaro belt from Pilot Knob (in California, just west of Yuma, Arizona) north to above Picacho State Recreation Area. The observed nests, found in cottonwood, willows (*Salix* spp.), and saguaros, were attributed to gilded flickers, as they were too large for Gila woodpeckers. Rosenberg et al. (1991) found gilded flickers at the Bill Williams River Delta, nesting almost exclusively in saguaros, although commonly foraging in the riparian forest. McKernan and Braden (2002) located gilded flickers during southwestern willow flycatcher (*Empidonax traillii extimus*) surveys along the LCR, from Topock Marsh south to Adobe Lake. The Arizona Breeding Bird Atlas recorded gilded flickers in Cibola NWR and Imperial NWR (Corman 2005).

Life History

General Description

The gilded flicker is a large woodpecker, measuring 28-31 cm in length and weighing 111 g. The breast and abdomen are beige, spotted heavily with black. The back is pale brown with black horizontal stripes. The crown is a rufous yellow color, contrasting with the gray throat and ear covert. The ventral side of the tail and wings are yellow. In flight, the white rump is obvious. The male has a red malar stripe, which is lacking in the female. The gilded flicker can only be confused with other flickers. The gilded flicker has the pale brown head of a red-shafted flicker but the yellow wings and tail of the yellow-shafted flicker.

Vocalizations and Sounds

Flickers produce an array of sounds, all for specific situations. Both sexes produce all vocalizations. The gilded and northern flickers' vocalizations are essentially identical, with the gilded flicker averaging a higher pitch (Sibley 2000). Flicker vocalization research is mostly associated with the northern flicker. Because the gilded and northern flickers' vocalizations are essentially the same, most of the following research on vocalizations and sounds are based on the northern flicker (Sibley 2000).

In the nest, hatchlings develop a buzzing vocalization (similar to a swarm of bees) that persists while in the nest. The hatchlings produce the buzzing sound as the nest cavity darkens while the parents enter the cavity. The hatchlings, hungry for food, crane their heads with their mouth wide open, producing the buzzing sound. A *Peah* vocalization develops just before fledging. The *Peah* is a single note lasting less than a second. Hatchlings give this call almost incessantly a few days before fledging. Duncan (1990 *in* Moore 1995) suggests the incessant calling is a method used by the adults to recognize their fledgling young.

The most common call produced by the adult is the Long Call, described as *wik-wik-wik*, *kick-kick-kick*, and *wick-a*, *wick-a*, *wick-a*. Lasting an average of 5 seconds, flickers typically produce the Long Call in the spring, during pair formation and territory establishment, and continue this vocalization into fall migration (Duncan 1990 *in* Moore 1995, Short 1982).

The adults also produce the single note *Peah* vocalizations given by the fledgling. The function of the *Peah* is unclear. Short (1972, 1982) and Kilhman (1983) consider the *Peah* an alarm call, but Lawrence (1966), Burns (1900), and Duncan (1990, *in* Moore 1995) suggest it is used to maintain contact between mates and or between parents and offspring. Moore (1995) suggests the *Peah* is a self-announcing call, not an alarm call.

The *Wicka* call described as *wik-a*, *wik-a*, *wik-a…*, and *ta-week*, *ta-week*, *ta-week…*, is given in unison by adult pairs, trios, and quartets engaged in close territorial and courtship "dances" (Short 1972, 1982). This call is complex, variable, and poorly understood.

The adults also make non-vocal drumming and tapping sounds with their bills. Flickers produce the drumming with rapid, even blows of the bill on a resonating object. Drumming usually occurs in conjunction with the Long Call as territorial defense (Lawrence 1966). Tapping sounds are associated with nest excavation and food gathering (Lawrence 1966).

Migration

Current literature suggests that the gilded flicker does not migrate (Hunter 1984, Rosenberg et al. 1991).

Habitat

In most of its breeding range, the gilded flicker is associated with mature saguaro cactus (*Carnegiea gigantae*; Gilman 1915, Bent 1939). Along the LCR, gilded flickers are associated with cottonwood-willow riparian areas (Gilman 1909, Grinnell 1914, Gilman 1915, Hunter 1984, McKernan and Braden 2002). Typically, gilded flickers stay away from densely populated urban and rural neighborhoods (Rosenberg et al. 1991, Corman 2005).

Diet

There are no detailed studies on the gilded flicker's diet. Grinnell (1914) identified black ants and ant larvae in gilded flicker gullets. Gilman (1915) observed gilded flickers eating cactus fruits and ants. Moore (1995) suggests that gilded flicker diet is similar to northern flickers. Beal (1911) reports northern flicker diets consisting mostly of ants (*Formica*, *Lasius*, *Campontus*, *Myrmica*, *Cremastogaster*, *Aphaenogaster*, *Prenolepis*, *Pheidole*, *Solenopsis*, *Tetramorium*) and ground beetles (*Carabidae*). Beal (1911) found that in the fall and winter flicker diets shifted to fruits. Primarily, flickers forage for food on the ground, in soil, and in anthills, probing and hammering their bill. Flickers are rarely found foraging in trees.

Behavior

Gilded flickers fly with an undulating trace, typical of all woodpeckers. Adult flight consists of a burst of wing flapping, alternating with non-flapping phases with wings folded against the body. Bent (1939) observed flickers running short distances and hopping while foraging on the ground. Flickers preen for extended periods, especially in the late afternoon and evening (Kilham 1983, Moore 1995). Their preening is typical of most passerines.

Flickers sleep clinging to a vertical surface, with their head tucked under their scapular feathers (Burns 1900, Moore 1995). Royall and Bray (1980), using radio transmitters to monitor roosting habitats, found flickers arriving at the roost just before sunset and leaving the roost up to 25 minutes before sunrise. They observed some flickers using the same roost every night and other flickers utilizing multiple roosts throughout the 2-month study.

Both sexes of flickers defend territories and mates aggressively, with a ritualized dance. Two birds of the same sex pair off using their bill as weapons against each other. Often a member of the opposite sex is watching the dance. The interactions can involve wing flapping, *Wicka* calling, and head and body bobbing. These interactions are common prior to breeding and are used for territory establishment, pair formation, and nest site selection (Short 1982, Moore 1995).

Male and female flickers appear to defend nesting territory; however, no detailed studies exist. Lawrence (1966) states that woodpeckers defend a small area around the nest tree, and have a territorial range with flexible boundaries, overlapping with neighboring woodpeckers.

Threats

Records exist of Harris hawk (*Parabuteo unicinctus*), sharp-shinned hawk (*Accipiter striatus*), Cooper's hawk (*A. cooperii*), and broad-winged hawk (*Buteo platypterus*) depredating flickers (Burns 1900, Miller 1925, Bent 1939). Various species of *Squamata*, *Rodentia*, and *Corvidea*, as well as raccoons (*Procyon lotor*), are common predators of flicker nestlings (Moore 1995). Flicker response to predators is rare (Moore 1995).

European starlings are common nest cavity competitors with flickers (Bent 1939, Ingold 1994, 1996); however, they may not compete with gilded flickers (Kerpez and Smith 1990). Kerpez and Smith (1990) studied competition between flickers and European starlings for nest cavities in saguaros around Tucson, Arizona. They found that European starlings do not compete with flickers. They attribute this to the larger size of flickers and the flickers ability to displace European starlings. Ingold (1994) studied nest competition between northern flickers and European starlings in Ohio. He found that European starlings seized 14% of the northern flickers nests. Ingold observed a European starling on the back of a northern flicker, pecking the flicker repeatedly.

Breeding

As gilded flickers are a non-migratory species, breeding behavior probably begins early in the season. Howell and van Rossem (1915) observed a red-shafted flicker and a gilded flicker "going through elaborate courting antics" in the Potholes region of the LCR in January. Corman (2005) reports gilded flicker courtship activity beginning in February. Nest cavity excavation may begin months before breeding, especially in saguaro trees as they need to "heal" before they are used (Corman 2005). Throughout the LCR MSCP area, gilded flickers utilize cottonwood, willow, saguaro, and honey mesquite (*Prosopis glandulosa*) trees for nest cavities (Grinnell 1914, Gilman 1915, Rosenberg et al. 1991).

Nest cavity construction is performed by both male and female flickers, with the male taking a dominate role (Lawrence 1966, Kilham 1983). Nest cavity construction can take weeks to complete (Lawrence 1966). Nest cavity dimensions along the LCR are poorly understood. Kerpez and Smith (1990) analyzed northern flicker (probably gilded flickers) saguaro nests in Tucson, Arizona. Nest cavity height averaged 6.2 m, entrance horizontal diameter averaged 8.3 cm, entrance vertical diameter averaged 7.0 cm, vertical depth averaged 37.6 cm, and horizontal depth averaged 12.5 cm. Nest cavity orientation is north-northeast (Zwatres and Nordell 1998). Zwatres and Nordell (1998) assumed that any hole meeting the stated dimension requirements was a gilded flicker nest cavity. They did not actually observe gilded flickers utilizing the holes studied. There is no nest microclimate data.

Corman (2005) reported gilded flickers occupying nests as early as 12 March, with young observed on 3 April. The latest observed young in a nest was 2 July (Corman 2005). Gilded flicker clutch size averaged 4.2 eggs per attempt, less than the 6.2 eggs per attempt observed for all other *Colaptes* species (Koening 1984). Moore (1995) reports that flickers produce only one brood per season but will re-nest if first attempt fails. Rosenberg et al. (1991) observed two broods in a season, with young in May and June.

Both flicker parents provide parental care during all phases of the nest cycle. Incubation usually takes 11 days. The nestlings fledge between 21 and 27 days old. Adult flickers may decrease feeding the young, and utilize *Peah* and Long Calls to entice hatchlings to leave the nest. Juvenile dependence on the adults is unknown but presumed to be short (Moore 1995).

Demography and Populations

First breeding for flicker males occurs the spring following birth. First breeding for females is unknown, but likely occurs during the spring following birth (Moore 1995).

Flicker abundance along the LCR is difficult to determine. Root (1988) utilized national Christmas Bird Count Data from 1963 to 1972 to determine winter abundance of flickers. Gilded flicker abundance was recorded at 1.6/h (individual birds observed/hour), less than the 4.72/h recorded for red-shafted flickers and the 2.77/h recorded for yellow-shafted flickers. Breeding Bird Survey (BBS) data from 1966 to 2005 show that national gilded flicker population trends decreased 0.85% (Sauer et al. 2005). The majority of BBS routes that have gilded flicker data occur outside of the LCR in southern Arizona. Therefore, due to the low number of BBS routes along the LCR, accurate population trends cannot be determined along the LCR.

Flickers can live at least 9 years and 2 months, based on banding records (Clapp et al. 1983). Survivorship records do not exist.

Nest Box/Snag Installation

There are no data on gilded flicker utilizing installed nest boxes or snags. Data on nest dimensions are limited. Kerpez and Smith (1990) analyzed northern flicker (probably gilded flickers) saguaro nests in Tucson, Arizona. Nest cavity height averaged 6.2 m, entrance horizontal diameter averaged 8.3 cm, entrance vertical diameter averaged 7.0 cm, vertical depth averaged 37.6 cm, and horizontal depth averaged 12.5 cm.

Conservation and Management

The impact of nest competition between gilded flickers and European starlings is still in question. The loss of nesting cavities due to competition and riparian habitat degradation and loss, particularly along the LCR, has had a negative effect on gilded flickers. These two issues are the biggest management concerns for gilded flickers.

LCR MSCP Conservation Measures

MRM3 – Conduct research to determine and access the effects of the nest site competition with European starlings on reproduction of covered species.
Research will be undertaken to determine whether nest site competition with European starlings is a substantial factor limiting the reproductive success of the elf owl, gilded flicker, and Gila woodpecker. If so, experimental programs may be implemented to determine the effectiveness and practicality of controlling starlings.

GIFL1 – Create 4,050 acres of gilded flicker habitat.
Of the 5,940 acres of created cottonwood-willow, at least 4,050 acres will be designed and created to provide habitat for this species. The 4,050 acres of habitat for the yellow-billed cuckoo will also provide habitat for the gilded flicker. The created habitat will be established in patches as large as possible but will not be created in patches smaller than 25 acres. In addition to the spatial replacement of affected habitat, the quality of created habitat will be substantially greater than affected habitat. Created habitat will be dominated by native riparian trees (i.e., cottonwood and willow trees), supporting a tree structure corresponding to structural type I-III.

GIFL2 – Install artificial snags to provide gilded flicker nest sites.
Until vegetation in created patches of gilded flicker habitat has matured sufficiently to support structural characteristics of nesting habitat (i.e., snags), install artificial snags that can be used by gilded flickers to excavate nesting cavities (LCR MSCP 2004).

LCR MSCP Research and Monitoring Needs

Gilded flicker populations have not been adequately studied along the LCR. Distribution, population status, demographics, habitat requirements, and threats have not been determined. Distribution and population status will be determined through multi-species system monitoring surveys. Habitat requirements need to be determined, including vegetation composition, structure, minimum patch size, density, and effects of habitat fragmentation. Gilded flickers are year-round residents; thus, seasonal movements and winter habitat requirements must be determined. Interactions between gilded flickers and other cavity nesting species, such as European starlings, need to be determined. Nest box design and use also need to be determined.

Literature Cited

Beal, F.E.L. 1911. Food of the woodpeckers of the United States. U. S. Department of Agriculture Biological Survey Bulletin 37.

Bent, A.C. 1939. Life histories of North American woodpeckers. U. S. National Museum Bulletin 174.

Burns, F.L. 1900. Monograph of the flicker. The Wilson Bulletin 7:1-82.

California Department of Fish and Game. 1988. Endangered and threatened animals of California. Habitat Conservation Planning Branch, California Department of Fish and Game, Sacramento.

Clapp, R.B., M.K. Klimiewicz, and A.G. Futcher. 1983. Longevity records of North American birds: Columbidae through Paridea. Journal of Field Ornithology 54:123-137.

Corman, T.E. 2005. Gilded Flicker. Pages 292-293 *In:* Corman, T. E., and C. Wise-Gervais (editors), Arizona Breeding Bird Atlas. University of New Mexico Press, Albuquerque.

Duncan, S. 1990. Auditory communication in breeding northern flickers (*Colaptes auratus*). Ph.D. Dissertation, University of Wisconsin, Milwaukee.

Gilman, M.R. 1909. Some owls along the Gila River in Arizona. The Condor 11:45-150

Gilman, M.F. 1915. Woodpeckers of the Arizona woodlands. The Condor 17:151-163.

Grinnell, J. 1914. An account of the mammals and birds of the lower Colorado valley. University of California Publications in Zoology 12:51-294.

Howell, A.B., and A. van Rossem. 1915. Additional observation on the birds of the lower Colorado valley in California. The Condor 17(6):November-December.

Hunter, W.C. 1984. Status of nine bird species of special concern along the Colorado River. Nongame Wildlife Investigations, Wildlife Management Ingold, Branch, Administrative Report 84-2, California Fish and Game Department, Sacramento, California. 63 p.

Ingold, D. 1994. Influence of nest-site competition between European starlings and woodpeckers. The Wilson Bulletin 106(2):227-241.

Ingold, D. 1996. Delayed nesting decreases reproduction success in northern flickers: Implications for competition with European starlings. Journal of Field Ornithology 67(2):321-326.

Kerpez, T.A., and N.S. Smith. 1990. Competition between European starlings and native woodpeckers for nest cavities in saguaros. The Auk 107:367-375.

Kilham, L. 1983. Life history studies of woodpeckers of eastern North America. Bulletin of the Nuttall Ornithology Club No. 20.

Koening, W.D. 1984. Clutch size of the gilded flicker. The Condor 86:89-90.

Lawrence, L.D. 1966. A comparative life-history study of four species of woodpeckers. Ornithological Monographs 5.

Lower Colorado River Multi-Species Conservation Program. 2004. Lower Colorado River Multi-Species Conservation Program, Volume II: Habitat Conservation Plan. Final. December 17. (J&S 00450.00.) Sacramento, California.

McKernan, R.L., and G. Branden. 2002. Status, distribution, and habitat affinities of the southwestern willow flycatcher along the lower Colorado River Year 6 – 2001. Submitted to Bureau of Reclamation, Lower Colorado River Region, Boulder City, Nevada, and USFWS, Carlsbad Field Office, California, and USFWS Reno Office, NV.

Miller, L. 1925. Food of the Harris hawk. The Condor 27(2):March-April.

Miller, L., and I. Demay. 1942. The fossil birds of California. University of California Publication Zoology 47:47-142.

Moore, W.S. 1995. Northern flicker (*Colaptes auratus*). *In:* A. Poole and F. Gill (editors), The Birds of North America, No. 413. The Birds of North America, Inc. Philadelphia, Pennsylvania.

Root, T. 1988. Atlas of wintering North American birds: An analysis of Christmas Bird Count data. University of Chicago Press, Chicago.

Rosenberg, K.V., R.D. Ohmart, W.C. Hunter, and B.W. Anderson. 1991. Birds of the lower Colorado River valley. University of Arizona Press, Tucson.

Royall, W.C. Jr., and O.E. Bray. 1980. A study of radio-equipped flickers. North American Bird Bander 5(2):47-50.

Sauer, J.R., J.E. Hines, and J. Fallon. 2005. The North American Breeding Bird Survey, results and analysis 1966-2005. Version 6.2. 2006. USGS Patuxent Wildlife Research Center, Laurel, Maryland.

Short, L.L. 1972. Systematics and behavior of South American flickers (Aves, *Colaptes*). Bulletin American Museum Natural History 149:1-109.

Short, L.L. 1982. Woodpeckers of the world. Delaware Museum Natural History Monograph, Serial 4.

Sibley, D.A. 2000. National Audubon Society, The Sibley Guide to Birds. Chanticleer Press, Inc. New York.

Zwartjes P.W., and S.E. Nordell. 1998. Patterns of cavity-entrance orientation by gilded flickers (*Colaptes chrysoides*) in cardon cactus. The Auk 115(1):119-126.

GILA WOODPECKER
(*Melanerpes uropygialis*)

Legal Status

The Gila woodpecker is not a Federally listed species. The state of California listed the Gila woodpecker as endangered in 1980 (CDFG 1980). The Gila woodpecker is not listed in either Arizona or Nevada.

Distribution

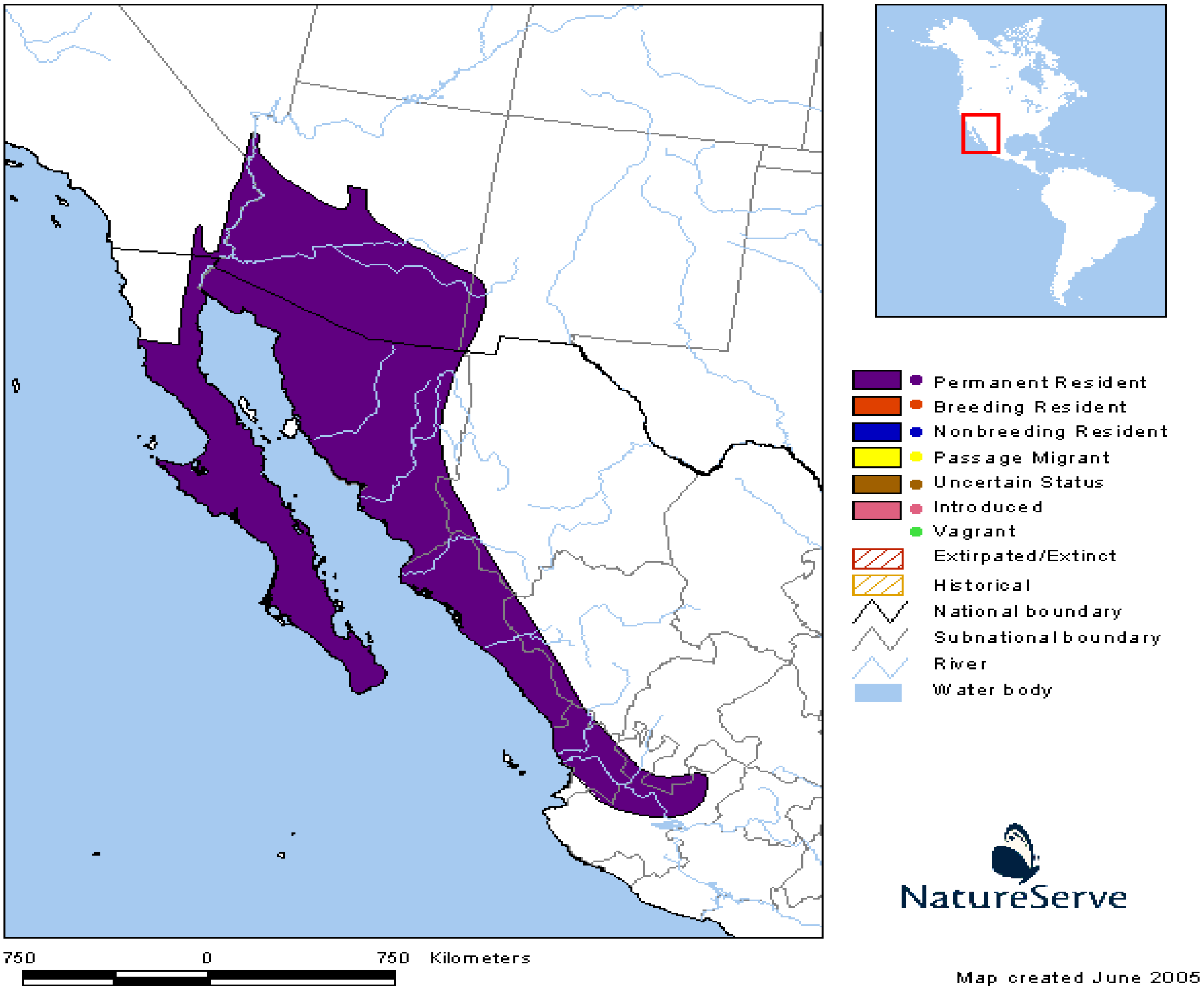
Historical Range

There are no known fossil records of the Gila woodpecker. Historically, the Gila woodpecker was found throughout southeastern California, along the lower Colorado River (LCR), and in extreme southern Nevada, southern Arizona, Mexico west to Baja California, south to Jalisco, and east to Chihuahua and Durango (Grinnell 1914, Hoffman 1927, van Rossem 1933, Grinnell and Miller 1944, Alcorn 1988, Rosenberg et al. 1991, Howell and Webb 1995).

Current Range

Currently, the Gila woodpecker has declined in southeastern California from its historical range. This decline is associated with the loss of woodland habitat and competition with European starlings (Garrett and Dunn 1981, Hunter 1984, Rosenberg et al. 1991, Kaufman 1996). The Gila woodpecker still occupies its historic range along the LCR, in extreme southern Nevada, southern Arizona, and throughout western Mexico. A few rare sightings outside of the Gila woodpecker's range have been documented. Hubbard (1978) reported Gila woodpeckers in extreme southwestern New Mexico, Willett (1933) reported individuals in Los Angles County, and Garrett and Dunn (1981) individuals in Ontario, California. Figure 1 illustrates the Gila woodpecker's current range.

Figure 1. Populations within LCR MSCP Project Boundary



Grinnell (1914) found the Gila woodpecker “at every station on both sides of the river” during his trip down the Colorado River. Coues (1866) reported the Gila woodpecker abundant along the LCR. More recently, Hunter (1984) reported 200 breeding individuals in California, with about half on private lands and in parks. Along the LCR, the Gila woodpecker can be found as far north as Clark County, Nevada, and south to Yuma, Arizona (Hollister 1908, LCR MSCP 2006a, 2006b). Rosenberg et al. (1991) suggested a decline in Gila woodpecker abundance along the LCR, reporting 650 individuals in 1976, 600 in 1983, and 561 in 1986, all on the Arizona side of the river, and estimated 1000 Gila woodpeckers along the whole stretch of the LCR. Breeding Bird Survey (Sauer et al. 2005) data provide an estimated population trend decline ranging from −1.5% to −3.4% from 1966 to 2004.

Life History

General Description

The Gila woodpecker is a noisy, aggressive, and conspicuous medium-sized woodpecker. Adults weigh 51-79 grams and measure 24 cm from head to tail. Adults have a light grayish-brown head, neck, and underparts. The bill is dull black. The abdomen is golden yellow and the legs are a dark greenish-brown. The back and tail are narrowly barred black and white. In flight, a white patch on the wings is obvious. At close distance, the male has a conspicuous red patch on the top of the head. Females and juveniles resemble the male; however, they lack the red patch on the head. Dimorphic in size, the male is 14% heavier, with a 14% longer bill.

Vocalizations

Categorized as loud and aggressive, the Gila woodpecker regularly produces two distinct calls. Bendire (1895) and Gilman (1915) described call 1 as the "sociable" call and call 2 a shrill "belly-aching" call. Call 1 consists of uniform vibrato notes with a simple temporal but rich harmonic structure. The call is similar to that of the northern flicker (*Colaptes auritus*) and the *Cha-aa-ah* call of the red-bellied woodpecker (*Melanerpes erythrocephalus*). Based on Brenowitz (1978a), males gave call 1 more than females (458 versus 78). The number of notes in a sequence is highly variable (mean 4.8 ± 4.8, n = 48). Duration of notes ranged from 0.18 to 0.95 seconds. Gila woodpeckers used call 1 primarily as interspecies territorial advertisement and secondarily as a display of the species.

Call 2 consists of a series of sharp *pip, pip* notes with complex harmonic structure. Males gave this call less often than females (48 versus 109). The number of notes in a sequence was variable (mean 6.8 ± 5.9 SD, n = 29). Intervals between the notes were 0.08 to 0.14 seconds. Gila woodpeckers gave this call in response to human disturbance and following vocalizations by other birds. Call 2 appears to be used as a general alarm call sometimes used in conjunction with visual displays in agnostic behaviors (Brenowitz 1978a).

Brenowitz (1978a) describes an infrequently used Gravel Call, spectrographically similar to the notes of Call 1; however, the call was shorter in duration and with an emphasis of different harmonics. This call was heard only when the Gila woodpecker was greatly agitated. A final vocalization, called a Combination Call, consisted of a note similar to the notes of Call 1, followed by several notes similar to the notes of Call 2. Brenowitz (1978a) heard this call only twice, by males being harassed by humans. He described it as an example of "ambivalent behavior" containing components of conflicting tendencies (self-advertisement and alarm).

Migration

A non-migratory species, the Gila woodpecker may roam locally short distances in the winter as food sources move (Kaufman 1996).

Habitat

Found in the arid deserts of the southwestern United States and northeastern Mexico, the Gila woodpecker utilizes saguaro cacti (*Carnegiea gigantea*) and riparian woodlands (Gilman 1915, Bancroft 1929). Within the LCR MSCP area, the Gila woodpecker is found along the river and washes in cottonwood-willow (*Populus* spp., *Salix* spp.) habitat. Gila woodpeckers have been found at a cottonwood-willow restoration site in Yuma, Arizona, and at a 20-year-old cottonwood-planting site (currently an LCR MSCP bird-banding site) at Havasu National Wildlife Refuge (LCR MSCP 2006a, 2006b). Hunter (1984) "found a strong relationship between Gila woodpeckers and high foliage density and diversity, and high foliage density and diversity coupled with high numbers of cottonwoods and willows." Hunter (1984) found that Gila woodpeckers utilized habitats that Anderson and Ohmart (1984) defined as cottonwood-willow I, II, and III.

Diet

The main food sources for the Gila woodpecker are insects such as ants (*Formicidae*), beetles (*Coleoptera*), grasshoppers (*Orthoptera*), cicadas (*Cicadidae*), and moths and butterflies (*Lepidoptera*). The fruits of the saguaros, and berries from mistletoe (*Phoradendron* spp.) and lyceum (*Lycium* spp.) are consumed as well (Edwards and Schnell 2000). Gilman (1915) observed Gila woodpeckers at feeding stations eating suet, meat, corn, peaches, pears, and eggs from chicken coops. MacRoberts and MacRoberts (1985) noted Gila woodpeckers storing oak (*Quecrcus* spp.) acorns. In cottonwood trees, Gila woodpeckers eat galls (egg sacs) from aphids (*Pemphigus populitransversus*) laid on the petioles of the leaves (Speich and Radke 1975). Gila woodpeckers will drink water from a container and sugar water from hummingbird feeders (Gilman 1915, Kaufman 1996).

Gila woodpeckers forage primarily on the trunk and inner branches of trees, probing for insects. Males were found to forage 60% of the time on the trunks of trees while females divided their time equally throughout the plant (Martindale 1984). Other foraging methods include pecking, probing, and gleaning (Edwards and Schnell 2000).

Within the LCR MSCP area, Anderson et al. (1982) found 4% plant and 96% animal material in the gizzards of Gila woodpeckers (n = 17) collected from March to October, and 100% animal material in the gizzards collected from November to March. Rosenberg et al. (1991), in studying the stomach contents of 15 Gila woodpeckers in summer riparian LCR habitats, found cicadas (>50%), ants, termites, beetles, insect larvae, and a few cactus fruits in their stomachs.

Behavior

Gila woodpecker flight is typical of woodpeckers: undulating, quick flapping bursts, alternating with short glides. They move up and down the trunks of trees searching for prey items and go to the ground for food, but no records exist of walking or hopping.

Gilman (1915) and Bent (1939) described Gila woodpeckers as highly aggressive towards both con- and hetero-specifics. In Brenowitz (1978b), Gila woodpeckers exhibited agnostic behavior toward other Gila woodpeckers, common flickers (*Colaptes* *auritus*), and European starlings (*Sturnus vulgaris*) during breeding. The agnostic behaviors included supplanting, chasing, and attacking with the bill. The most aggressive behaviors were related to defense of the nest. Martindale (1982) observed mated pairs working as a team to deliver food to the nest and defend the nest simultaneously. Males were more aggressive than females, attacking intruders more frequently with greater zeal. Females tended to use only agnostic vocalizations. Males were able to drive off males and females, while females were generally unable to drive off males, presumably due to the degree of size dimorphism. Brenowitz (1978b) used stuffed decoys to observe reactions and found only same-sex decoys were attacked.

Territory sizes range from 4.45 to 10.00 ha (Hensley 1954). Gila woodpeckers in washes had smaller territories while larger territories were established in the open desert areas.

Breeding

The earliest report of nest excavation is in February, when pairing and territorial chasing was first evident (Rosenberg et al. 1991, Bradley 2005).

The height of Gila woodpecker nesting season is mid-April through mid-May (Gilman 1915, Bradley 2005). Along the LCR, fledglings have been seen as early as April and as late as July (Anderson et al. 1982, Rosenberg et al. 1982, Bradley 2005). Gila woodpeckers sometimes lay second and third clutches (Phillips et al. 1964, Inouye et al. 1981). In the LCR valley, Rosenberg et al. (1991) observed family groups with first brood offspring remaining as the adults attended second nests.

Nest cavity competition exists with elf owls (*Micrathene whitneyi*) and European starlings, with both species documented evicting Gila woodpeckers from the woodpecker's cavities (Gilman 1915, Brush 1984). Most breeding data comes from saguaro habitat, although Gila woodpeckers will nest in cottonwood, willow, sycamore (*Plantus* spp.), and ash (*Fraxinus* spp.) in riparian areas, and palm (*Washingtonia* spp.), eucalyptus (*Eucalyptus* spp.), Athel tamarisk (*Tamarix aphylla*), and mulberry trees (*Morus* spp.) in urban areas (Bradley 2005, Rosenberg et al. 1991). Korlo and Hutto (1984) reported Gila woodpeckers disproportionately utilizing taller saguaros with nonrandom oriented cavity entrances. Inouye et al. (1981) also report nonrandom oriented saguaro cavities with the mean direction north-northwest. Kerpez and Smith (1990a) concluded that Gila woodpecker saguaro nest cavities were randomly oriented. Kerpez and Smith (1990a) notes that Inouye et al. (1981) and Korlo and Hutto (1984) recorded all cavities observed regardless of breeding evidence, while Kerpez and Smith (1990a) only measured known breeding cavities.

Demography and Populations

No data is available for age at first breeding and lifetime reproductive success. Gila woodpeckers generally rear two or three broods per season (Bent 1939, Phillips et al. 1964, Inouye et al. 1981).

Along the California side of the LCR, Hunter (1984) estimated the total population of Gila woodpeckers to be 200 individuals from surveys conducted during 1975-1979.

Rosenberg et al. (1991) estimated 1,000 Gila woodpeckers along the whole stretch of the LCR. Both Hunter (1984) and Rosenberg et al. (1991) lack any information on how they acquired their population estimates. Breeding Bird Survey (Sauer et al. 2005) data estimate a population trend decline ranging from −1.5% to −3.4% from 1966 to 2004.

Nest Box/Snag Installation

Gila woodpecker nest cavities in cottonwood, willow, and mesquite (*Prosopis* spp.) trees are not well documented in the literature and no data exist on Gila woodpecker's utilizing nest boxes or artificial snags. Brush (1983) has the only data on cavity dimensions, recording mean cavity entrance diameters of 5.4 cm (n = 7) and cavity depths of 26.4 cm (n = 5). Mean cavity heights of 1.5 m (n = 6) in mesquite and 10.3 m (n = 16) in cottonwoods and willows were recorded.

Conservation and Management

In the Sonoran Desert, European starlings and human development threaten Gila woodpeckers (Kerpez and Smith 1990a, 1990b). Gila woodpeckers compete with European starlings for nesting sites. In the Southwest, European starling numbers have more than doubled in the past 30 years (Edwards and Schnell 2000). Near Blythe, California, Hunter (1984) reported European starlings removing a nesting pair of Gila woodpeckers from three different nests and removing the woodpecker's eggs once. Near Parker Dam, Arizona, Hunter (1984) observed European starlings displacing two pairs of Gila woodpeckers. Brush (1983) observed three pairs of European starlings displace Gila woodpeckers from cavities near the Bill Williams River delta.

Along the LCR, the loss of cottonwood-willow stands has reduced the numbers of Gila woodpeckers (Rosenberg et al. 1991). Rosenberg et al. (1991) found that cottonwood-willow stands less than 20 ha in area were devoid of Gila woodpeckers. Tweit and Tweit (1986) showed that urban residential housing development, at a density of 2 houses/ha, did not reduce the number of Gila woodpecker, provided native vegetation was maintained.

LCR MSCP Conservation Measures

MRM3 – Conduct research to determine and assess the effects of nest site competition with European starlings on reproduction of covered species.
Research will be undertaken to determine whether nest site competition with European starlings is a substantial factor limiting the reproductive success of the elf owl, gilded flicker, and Gila woodpecker. If so, experimental programs may be implemented to determine the effectiveness and practicality of controlling starlings.

GIWO1 – Create 1,702 acres of Gila Woodpecker habitat.
Of the 5,940 acres of created cottonwood-willow habitat, at least 1,702 acres will be designed and created to provide habitat for this species in reaches 3-6. Patches of created habitat will be designed and managed to support cottonwood-willow types I-IV in patches as large as possible, but will not be created in patches smaller than 50 acres to achieve, based on the best available information, the minimum habitat patch size requirements of the species. In addition to the spatial replacement of affected habitat, the quality of created habitat will be substantially greater than affected habitat. Created habitat, thus, will approximate the condition of native habitat of the species that was historically present along the LCR.

GIWO2 – Install artificial snags to provide Gila Woodpecker nest sites.
Until vegetation in created patches of Gila woodpecker habitat has matured sufficiently to support structural characteristics of nesting habitat (i.e., snags), install artificial snags that can be used by Gila woodpeckers to excavate nesting cavities (LCR MSCP 2004).

LCR MSCP Research and Monitoring Needs

Gila woodpecker populations have not been adequately studied along the LCR. Distribution, population status, demographics, habitat requirements, and threats have not been determined. Distribution and population status will be determined through multi-species system monitoring surveys. Habitat requirements need to be determined, including vegetation composition, structure, minimum patch size, density, and effects of habitat fragmentation. Gila woodpeckers are year-round residents; thus, seasonal movements and winter habitat requirements must be determined. Interactions between Gila woodpeckers and other cavity nesting species, such as European starlings, need to be quantified and remedial measures, if necessary, determined. Determining European starling demographics and habitat requirements may provide information for site selection of habitat creation sites. Nest box design and use need to be determined.

Literature Cited

Alcorn, J.R. 1988. The birds of Nevada. Fairview West Publishing. Fallon, Nevada.

Anderson, B.W., R.D. Ohmart, and S.D. Fretwell. 1982. Evidence for social regulation in some riparian bird populations. The American Naturalist 120:340-352.

Anderson, B.W., and R.D. Ohmart. 1984. Lower Colorado River riparian methods of quantifying vegetation communities to prepare type maps. Final Report. Bureau of Reclamation, Lower Colorado Region, Boulder City, Nevada.

Bancroft, G. 1929. The breeding birds of central lower California. The Condor 32:20-49.

Bendire, C.E. 1895. Life histories of North American birds. U.S. National Museum Special Bulletin 3.

Bent, A.C. 1939. Life histories of North American woodpeckers. Dover Publications, Inc. New York.

Bradley, B. 2005. Gilder Flicker. *In:* Corman, T.E. and C. Wise-Gervais (editors), Arizona Breeding Bird Atlas. University of New Mexico Press, Albuquerque. 636 p.

Brenowitz, G.L. 1978a. An analysis of Gila woodpecker vocalizations. The Wilson Bulletin 90:451-455.

Brenowitz, G.L. 1978b. Gila woodpecker agnostic behavior. The Auk 95:49-58.

Brush, T. 1983. Cavity use by secondary cavity-nesting birds and response to manipulations. The Condor 85:461-466.

California Department of Fish and Game. 1980. Endangered and threatened animals of California. Habitat Conservation Planning Branch, California Department of Fish and Game, Sacramento.

Coues, E. 1866. List of the birds of Fort Whipple, Arizona. Proceeding of the Academy of National Sciences. Philadelphia 18:39-140.

Edwards, H.H., and G.D. Schnell. 2000. Gila woodpecker (*Melanerpes uropygialis*). *In:* A. Poole and F. Gill (editors), The Birds of North America, No. 532. The Birds of North America, Inc., Philadelphia, Pennsylvania.

Garrett, K., and J. Dunn. 1981. Birds of southern California: Status and distribution. Los Angeles Audubon Society, Los Angeles, California.

Gilman, M.F. 1915. Woodpeckers of the Arizona woodlands. The Condor 17:151-163.

Grinnell, J. 1914. An account of the mammals and birds of the lower Colorado Valley. University of California Publications in Zoology 12:51-294.

Grinnell, J., and A.H. Miller. 1944. The distribution of the birds of California. Pacific Coast Avifauna 27:1-608.

Hensley, M.M. 1954. Ecological relations on the breeding bird population of the desert biome in Arizona. Ecological Monographs 24:185-207.

Hoffman, R. 1927. The Gila woodpecker at Holtville, Imperial County, California. The Condor 29:162.

Hollister, N. 1908. Birds of the region about Needles, California. The Auk 25:455-462.

Howell, S.N.G., and S. Webb. 1995. A guide to birds of Mexico and northern Central America. Oxford University Press, New York.

Hubbard, J.P. 1978. Revised check-list of the birds of New Mexico. New Mexico Ornithological Society Publication No. 6.

Hunter, W.C. 1984. Status of nine bird species of special concern along the Colorado River. Nongame Wildlife Investigations, Wildlife Management Branch, Administrative Report 84-2, California Fish and Game Department, Sacramento, California. 63 p.

Inouye, R.S., N.J. Huntly, and D.W. Inouye. 1981. Non-random orientation of Gila woodpecker nest entrances in saguaro cactus. The Condor 83:88-89.

Kaufman, K. 1996. Lives of North American birds. Houghton Mifflin Company, Boston, Massachusetts.

Kerpez T., and N.S. Smith. 1990a. Nest-site selection and nest-cavity characteristics of Gila woodpeckers and northern flickers. The Condor 92:193-198.

Kerpez T., and N.S. Smith. 1990b. Competition between European starlings and native woodpeckers for nest cavities in saguaros. The Auk 107:367-375.

Korol, J.J., and R.L. Hutto. 1984. Factors affecting nest site location in Gila woodpeckers. The Condor 86:73-78.

Lower Colorado River Multi-Species Conservation Program. 2004. Lower Colorado River Multi-Species Conservation Program, Volume II: Habitat Conservation Plan. Final. December 17. (J&S 00450.00.) Sacramento, California.

Lower Colorado River Multi-Species Conservation Program. 2006a. Avian post development monitoring at the Pratt Restoration Site breeding season 2005. Lower Colorado Region, Bureau of Reclamation, Boulder City, Nevada.

Lower Colorado River Multi-Species Conservation Program. 2006b. Final report for the operation of two Monitoring Avian Production and Survivorship (MAPS) stations on the lower Colorado River, 2005 Breeding Season. Lower Colorado Region, Bureau of Reclamation, Boulder City, Nevada.

MacRoberts, M.H., and B.R. MacRoberts. 1985. Gila woodpecker stores acorns. The Wilson Bulletin 97:571.

Martindale, S. 1982. Nest defense and central place foraging: A model and experiment. Behavioral Ecological Sociobiology 10:85-89.

Martindale, S. 1984. Sexual dimorphism and parental role switching in Gila woodpeckers. The Wilson Bulletin 96:116-121.

Phillips, A., J. Marshall, and G. Monson. 1964. The birds of Arizona. University of Arizona Press, Tucson.

Rosenberg, K.V., R.D. Ohmart, and B.W. Anderson. 1982. Community organization of riparian breeding birds: Response to an annual resource peak. The Auk 99:260-274.

Rosenberg, K.V., R.D. Ohmart, W.C. Hunter, and B.W. Anderson. 1991. Birds of the lower Colorado River valley. University of Arizona Press, Tucson.

Sauer, J.R., J.E. Hines, and J. Fallon. 2005. The North American Breeding Bird Survey, results and analysis 1966-2004. Version 2005.2. USGS Patuxent Wildlife Research Center, Laurel, Maryland.

Speich S., and W.J. Radke. 1975. Opportunistic feeding of the Gila woodpecker. The Wilson Bulletin 87:275-276.

Tweit, R.C., and J.C. Tweit. 1986. Urban development effects on the abundance of some common resident birds of the Tucson area of Arizona. American Birds 40:431-436.

van Rossem, A.J. 1933. The Gila woodpecker in the Imperial Valley of California. The Condor 35:74.

Willett, G. 1933. A revised list of the birds of southwestern California. Pacific Coast Avifauna 21:1-204.

VERMILION FLYCATCHER (*Pyrocephalus rubinus*)

Legal Status

The vermilion flycatcher is not currently Federally listed. In California, the flycatcher is considered a second priority species of special concern. The species of special concern list is divided into three categories: highest, second, and third priorities. Species in the highest priority category face immediate extirpation of their entire California population, or their California breeding population, if current trends continue. Species in the second priority category are on the decline in a large portion of their range in California, but their populations are still sufficiently substantial that extirpation is not imminent. Species in the third priority category are not in any present danger of extirpation and their populations, within most of their range, do not appear to be declining seriously; however, simply by virtue of their small populations in California, they are vulnerable to extirpation should a threat materialize.

Distribution

Comprising 12 subspecies, the vermilion flycatcher ranges from the southwestern United States, south to central Argentina and Uruguay (Figure 1) (AOU 1998, Wolf and Jones 2000). The subspecies found within the LCR MSCP boundary is *P. r. flammeus*, which ranges from south-central California, southern Nevada, southern Arizona, and southern Texas south to Baja California, Sonora, and Nayarit, Mexico (AOU 1957, Wolf and Jones 2000).

Historical Range

Vermilion flycatchers were considered numerous in the lower Colorado River Valley in the early part of the 1900s (Grinnell 1914, Rosenberg et al. 1991). Grinnell and Miller (1944) considered the vermilion flycatcher to be “fairly common” within the breeding range of the Colorado Desert, including the Imperial Valley, northwest to at least Coachella in the Coachella Valley, Riverside County, and the lower Colorado River Valley, from the Mexican border to north of Needles, California, with all known nesting localities below 150 m (500ft) elevation. Early in the 20th century, nesting was also recorded in the upper Mohave River drainage. Breeding birds persisted in the Coachella Valley to at least the late 1950s (Garrett and Dunn 1981), but the species no longer nests there (Patten et al. 2003); it also has declined in the Imperial Valley, where it is now considered a “rare” breeder (Patten et al. 2003).

Vermilion flycatchers have undergone a significant range shift in California during the past five decades. This change in status, from breeder to early winter visitor, has been documented in other parts of the flycatcher’s Sonoran Desert range (Rea 1983). The vermilion flycatcher is now virtually unknown as a breeder in the Sonoran Desert of

California (Rosenberg et al. 1991), but breeds in many locations in the Mojave Desert, almost all of which are well above 150 m (500 ft) elevation.

Serena (1981) lists locations where vermilion flycatchers have been found in California. These areas included the Blythe golf course and the Clark Ranch. The Clark Ranch had an unmated female during the summer of 1982. Hunter (1984) reported a few breeding pairs along the Bill Williams River, near Yuma, and near Willow Valley Estates (near Needles) in Arizona. During the 1983 summer season, only one breeding pair was found on the LCR mainstem at the Parker Dam residences on 21 April. This pair fledged two young by 9 June. No other individuals were found on this survey. During the winter, individuals, probably not all from Colorado River breeding populations, were found in the agricultural-riparian vegetation interface areas (Hunter 1984).

Vermilion flycatchers were described as common to abundant summer residents in mesquites, willows, and cottonwoods in southern and central Arizona, but rather local along the Salt and Colorado river valleys (Phillips et al. 1964).

Current Range

The vermilion flycatcher is locally common in southern Nevada. The only significant population of vermilion flycatchers in Nevada is found on the 1,200-acre Mormon Ranch in Moapa Valley. Surrounded by Mojave Desert scrub, the Moapa Valley accommodates the flycatcher's need of mesquite groves for nesting, and flooded fields for catching flying insects (Patten 2006).

The vermilion flycatcher breeds in Arizona from the northwest and Mogollon Rim, south throughout the state. The flycatcher is common along the base of the Huachuca Mountains, and absent from the southwestern corner of the state. Atlas data reveal local concentrations along major drainages such as the Gila, Salt, Verde, Agua Fria, Santa Cruz, and San Pedro rivers. Vermillion flycatchers were also found regularly to west-central Arizona along the Hassayampa, Bill Williams, Santa Maria, and Big Sandy river systems. Atlasers found vermilion flycatchers to be a local breeder along the lower Colorado River (Averill-Murray and Corman 2005). The flycatcher is reported as a rare and local breeder along the lower Verde, Salt, and lower Colorado rivers (Monson and Phillips 1981, Rosenberg et al. 1991). The flycatcher also breeds in southern New Mexico in the Pecos, San Francisco, Gila, and lower/middle Rio Grande valleys, with occasional summer records from northeastern New Mexico in San Miguel and Union counties (Hubbard 1978). The vermilion flycatcher breeds in western and central Texas, and occasionally breeds in central and western Oklahoma (Wolf and Jones 2000).

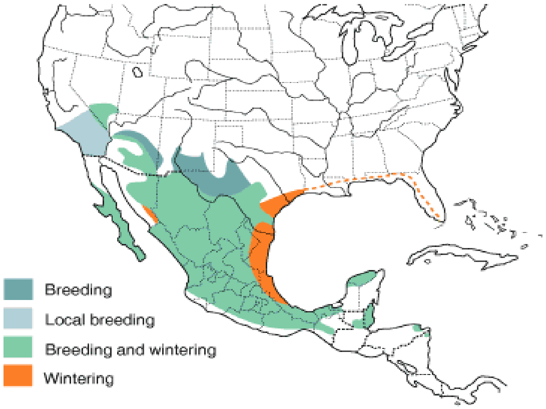
The breeding range has declined throughout southern California (Rosenberg et al. 1991). The vermilion flycatcher is currently a local breeder, restricted to a small number of individuals currently or recently breeding in the following locations in California: Yucca Valley, Apple Valley, Fort Irwin, Mojave River, Barstow, California State University-San Bernardino, Twenty-nine Palms, and Iron Mountain Pumping Plant in San Bernardino County; Ridgecrest, China Lake, and the South Fork of the Kern River Preserve in Kern County; Antelope Valley, Ridgecrest, California City, and Leona Valley in Los Angeles County; Lake Tamarisk in Desert Center in Riverside County; Santa Barbara Canyon and New Cuyama in Santa Barbara County; and Warner Valley, Santa Ysabel Asistincia, Sweetwater River, Anza-Borrego Desert, Borrego Springs, Mason Valley, and Vallecito Valley in San Diego County (Patten 2006, Unitt 2004).

Vermilion flycatchers are resident throughout all but the northernmost portion of their breeding range in the United States, Mexico, and Central America. This species winters outside of the breeding range in deserts of southeastern California and southwest Arizona. A few individuals winter regularly along the California coast, north to Ventura County, along the Gulf Coast (rarely north to southern Arkansas), and throughout the mainland of Florida (Wolf and Jones 2000). The winter range fluctuates with weather conditions. In some winters, the flycatcher will wander along river corridors outside its normal range (Grinnell and Miller 1944). Vermilion flycatchers winters where average minimum January temperature is usually above –1°C. Range extensions of this temperature gradient are along protected river valleys, including the Mississippi, Brazos, Pecos, Rio Grande, and Colorado rivers (Root 1988). Vermilion flycatchers were seen wintering in high numbers in south Texas, around Nogales, Arizona (Root 1988), and along the Colorado River regularly to Topock, Arizona (Phillips et al. 1964).

Populations within LCR MSCP Project Boundary

Significant populations occur at the Bill Williams River NWR, the Blythe golf course, Clark Ranch, the Parker Dam residences, and Willow Valley Estates (Rosenberg et al. 1991). This species has also been found recently in areas being restored with native cottonwood and willow. Single male vermilion flycatchers were detected wintering on the Cibola Nature Trail area searches from 2002 to 2004 (Reclamation unpubl. data). Flycatchers were also present during the breeding season in the mesquite parks at the Ahakav restoration area on the Colorado River Indian Tribe land south of Parker, Arizona (B. Sabin, pers. com.). During focused surveys for other riparian obligate species along the Colorado River in the late 1990s and early 2000s, biologists from the San Bernardino County Museum incidentally detected dozens of pairs of vermillion flycatchers between Parker Dam and the Mexican border (Patten 2006).

***Figure 1*. Distribution of the vermilion flycatcher. This species winters north and east locally to the dashed orange line (Wolf and Jones 2000).**



Life History

General Description

The vermilion flycatcher is a small flycatcher with a length about 13-14 cm, and a mass of 11-14 g. The plumage is sexually dichromatic. The adult male has bright vermilion on the top of the head with underparts bright vermilion, scarlet, or orange. The lores, ear-coverts, and nape form a dark blackish-brown mask, with the remaining upperparts including wings and tail colored blackish brown. The adult female has the top of the head, ear coverts, and remaining upperparts, including wings and tail, colored grayish brown, becoming darkest on the tail. The forehead and indistinct superciliary stripe are grayish white, with the remiges and wing coverts margined paler, forming wing bars on the greater and median coverts. The female's underparts are whitish, becoming pale red to salmon-colored toward the posterior, and finely streaked with gray on the breast, sides, and flanks. Adult plumages are similar throughout the year. The immature female is similar to the adult female, except posterior underparts are yellowish. Immature males show delayed plumage maturation, remaining similar to the adult female throughout summer of the second calendar year. Underparts of immature males are extensively covered with salmon pink or pale orange-red (Wolf and Jones 2000).

Breeding

Despite the vermilion flycatcher's conspicuousness, its biology remains poorly known (Wolf and Jones 2000). This species is mainly a summer visitor to the southwestern United States, although it regularly winters in the Sonoran and Chihuahuan deserts and in cismontane southern California. Breeding birds of the Colorado Desert are generally resident, but those in colder regions (such as the Mojave Desert) are migratory and withdraw at least partially to different habitats in winter (Grinnell and Miller 1944). Birds that migrate arrive on their breeding grounds by late February or early March and typically depart by late September (egg dates range from early March to early July; Bent, 1942). The breeding season extends from early March through early July (Rosenberg et al. 1991). In Arizona, males arrive on the breeding grounds first, beginning in early to mid-February. Nest construction can begin by late February. Atlasers confirmed breeding (nests with young) through early August, with a peak in activity between late April and late June (Averill-Murray and Corman 2005).

These birds are monogamous. The nest site is chosen based on the Nest-Site-Showing Display of the male. He flies around to potential nesting sites and gives a soliciting call to the nearby females, encouraging them to take a look. They fly to each site, crouch, and make nest forming movements while letting out a chatter call. They also flutter their wings during this display. The chosen nest site by the female is usually within 200 m of the male's preferred nest site. The female will often ignore the displaying male, but when she decides to accept, she and the male will land at different potential nest sites in a crouching position. They will display side by side. The male will retreat when he observes that the female is starting the nest construction. The construction begins almost immediately after the female chooses the site (Carothers 1974).

Vermilion flycatchers first breed as second-year birds, the first spring after hatching. Males usually arrive on the breeding grounds a week or so earlier than females, as early as February, and as late as the first week of April. The earliest nest observed was constructed in late March. The nests are loosely constructed and made of twigs, grasses, fibers, and empty cocoons, and lined with down, feathers, and hair. Nest shape is a shallow cup. The female completes the nest with cobwebs and lichens. Egg-laying occurs as soon as the nest has been finished. The clutch is usually made of 2-3 oval-shaped eggs. They range in color from pure white to cream, tan, or brown. The larger end of the egg is usually marked with a dark brown spot (Carothers 1974).

Immediately after the eggs are laid, the female begins incubation. All eggs hatch by 14 days in most cases, but the average length of time is 13-15 days. While the female incubates the eggs, the male feeds her. He lands on a nearby branch and announces himself with a contact call. He quickly deposits the food in the female's mouth and promptly leaves. During this period, copulations often occur on the nest. Females have never been observed begging for food. The male feeds the female on average every 1.5 hours. Following feeding, copulation is likely to occur. Females are extremely vigilant when they are at the nest. They are most alert in the early morning when the eggs are exposed to full sunlight. The female will often stand over the eggs so the sunlight cannot reach them. The female occasionally leaves the nest, but never goes very far. Young are altricial, weighting a little over a gram. Their eyes start to open about 4 days after hatching. There seems to be no correlation between the feeding rate and the number of young in the nest. Both parents feed the young, approximately 3.5 times per hour. They are fed mostly butterflies and moths. About half of their food is made of larval Lepidoptera. The female broods the young and they fledge approximately 13-15 days after hatching. Second broods are common. Second clutches have been observed from 20 May to 10 June (Carothers 1974).

Diet

The vermilion flycatcher prefers open areas and often perches in a conspicuous location from which it sallies frequently attempting to capture prey. No systematic studies of diet have been conducted, but like all flycatchers, this species consumes insects and other arthropods. Among the insects known to be taken are grasshoppers, beetles, flies, and bees (Bent 1942, Wolf and Jones 2000).

Ninety-four percent of foraging takes place within 3 m of the ground, with the least amount of foraging occurring over water. They are a sit-and-wait predator, sitting on perches and sallying down to catch single insects one at a time. Sometimes they carry captured prey to their perch and beat it before consuming it (Fitzpatrick 1980).

Habitat

In the breeding season, vermilion flycatchers occupy arid scrub, farmlands, savanna, agricultural areas, and riparian woodland. They are often associated with surface water, and in Arizona, occur where cottonwoods (*Populus* spp.), willows (*Salix* spp.), oaks (*Quercus* spp.), mesquites (*Prosopis* spp.), and sycamores (*Platanus* spp.) line streams (Wolf and Jones 2000).

When the vermilion flycatcher formerly bred in the Sonoran Desert of California, it was associated with low-lying, open riparian areas with accessible water (either pooled or flowing) and dominated by mesquite, willow, and Fremont cottonwood (Grinnell and Miller 1944). At some sites in California, such as Morongo Valley and Victorville, the flycatchers use cottonwood-willow woodland, but they also inhabit golf courses, residential areas, and parks (Garrett and Dunn 1981, Wolf and Jones 2000).

On the LCR, vermilion flycatchers are most often found in riparian woodland dominated by willows and cottonwoods with mesquites, surface water, and pastureland frequently nearby (Rosenberg et al. 1991). In Arizona, nests are usually placed in native trees such as Goodding's willows, Fremont cottonwoods, mesquites, Arizona walnuts, Arizona sycamores, desert willows, acacia, and paloverde, but sometimes in nonnative trees such as elms (*Ulmus* spp.), olives (*Olea europaea*), black locusts (*Robinia pseudoacacia*), saltcedar *(Tamarix chinensis),* and eucalyptus (*Eucalyptus* spp.), especially in parks or near human habitations (Rosenberg et al. 1991, Wolf and Jones 2000, Averill-Murray and Corman 2005). Arizona atlasers described substrates for 19 nests; 42% were found in mesquite (Averill-Murray and Corman 2005). Rangewide, vermilion flycatcher nests are placed in horizontal forks of trees at heights ranging from 1 to 18 m above ground (Rosenberg et al. 1991, Wolf and Jones 2000).

In central Arizona and northern Mexico, the vermilion flycatcher was not found in areas where Fremont cottonwoods form dense canopy and mesquite form thick understory. The flycatcher breeds in two distinct vegetative associations: broadleaf riparian woodland (cottonwood-dominated) and in the often adjacent microphyllous association (mesquite-dominated). The cottonwood association includes Goodding's willow (*Salix gooddingii;* relative frequency > 10%), Arizona ash (*Fraxinus velutina;* relative frequency < 3%), and rarely Arizona sycamore (*Platanus wrightii*) and Mexican elderberry (*Sambucus mexicanus*). Scattered scrubs include saltcedar (*Tamarix pentandra*), seep-willow (*Baccharis glutinosa*), and honey mesquite (*Prosopis juliflora*). The herbaceous community was composed primarily of bee-plant (*Cleome lutea*), sacred datura (*Datura meteloides*), nightshade (*Solanum elaeagnifolium*), and tall white nettle (*Urtica gracilis*). The ground cover was composed of Bermudagrass (*Cynodon dactylon;* 15-95%).

The flycatcher also breeds in mesquite bosques where honey mesquite was the dominant tree species. The following trees were found in lesser numbers: desert willow (*Chilopsis linearis;* <10%), Goodding's willow (<2%), Fremont cottonwood (<1%), saltcedar (<1%), and blue paloverde (*Cercidium floridum*). Scrub community was composed of honey mesquite, catclaw (*Acacia greggii*), seep-willow, wait-a-minute (*Mimosa buincifera*), wolf-berry (*Lysium pallidum*), and saltbush (*Atriplex* spp.). The total ground cover was 10-60%, dominated by annual plants and burrobrush (*Hymenoclea* spp.) with Bermudagrass found only occasionally (Carothers 1974). Vegetation data from this study did not include sites along the LCR.

Threats

The primary threat to the vermilion flycatcher in this portion of its range is loss of riparian woodlands. The destruction of much of the native riparian habitat along the LCR, and its replacement in many areas by nonnative saltcedar (*Tamarix* spp.), has probably led to its near-extirpation as a breeding species in this area (Rosenberg et al. 1991). The increase in flycatcher nesting locations in the Mojave Desert of California perhaps can be attributed to the advent of various man-made habitat oases, such as parks, golf courses, and suburban housing places, in areas formerly supporting desert scrub.

Brood parasitism by brown-headed cowbirds (*Molothrus ater*) may contribute slightly to population declines in California, but the vermilion flycatcher appears to be an uncommon host (Friedmann 1963, Friedmann et al. 1977, Friedmann and Kiff 1985). The parks and golf courses now frequently used by nesting vermilion flycatchers provide excellent foraging habitat for the cowbird.

LCR MSCP Conservation Measures

VEFL1 – Create 5,208 acres of vermilion flycatcher habitat.
Of the 7,260 acres of created cottonwood-willow and honey mesquite, at least 5,208 acres will be designed and created to provide habitat for this species. Patches of created habitat will be designed and managed to support cottonwood-willow types I-IV and honey mesquite type III that provide habitat for this species. The created habitat will be established in patches as large as possible. At a minimum, however, isolated patches of honey mesquite will be created in patches of at least 50 acres, and of the 5,940 acres of LCR MSCP-created cottonwood-willow, 1,702 acres will be created in patches of at least 50 acres, 2,348 acres will be created in patches of at least 25 acres, and 1,890 acres will be created in patches of at least 10 acres.

In addition to the spatial replacement of affected habitat, the quality of created habitat will be substantially greater than affected habitats. Patches of existing cottonwood-willow in the LCR MSCP planning area typically include dense stands of saltcedar that support little vegetative diversity relative to the cottonwood-willow land cover that will be created as habitat. Created habitat will be dominated by native riparian trees (i.e., cottonwood and willow trees), support a tree structure corresponding to structural types I-IV, support a diversity of plant species, and be created to the greatest extent practicable in patch sizes optimal for supporting the species. Created habitat, thus, will approximate the condition of the native habitat of the species that was historically present along the LCR. The design and management criteria described in the conservation measures for the southwestern willow flycatcher (Section 5.7.2) and yellow-billed cuckoo (Section 5.7.14) will ensure that created cottonwood-willow stands in structural types I-IV will also provide other habitat requirements for this species (e.g., habitat patch size, food requirements).

LCR MSCP Research and Monitoring Needs

The vermilion flycatcher is a year-round resident along the LCR. Comprehensive surveys need to be conducted to determine distribution and population status. Habitat requirements during breeding season and winter need to be determined, including vegetation composition, structure, microhabitat characteristics, minimum patch size, and prey availability. Demographic data, such as site tenacity, site fidelity, nest success, and productivity, are needed to design and manage created habitat.

Literature Cited

American Ornithologists’ Union (AOU). 1957. Check-list of North American birds. 5th ed. American Ornithologists’ Union, Washington, DC.

American Ornithologists' Union (AOU). 1998. Check-list of North American Birds, 7th ed. American Ornithologists’ Union, Washington, DC.

Averill-Murray and Corman, T.E. 2005. Vermilion flycatcher. *In:* Corman, T. E. and Wise-Gervais (editors), The Arizona Breeding Bird Atlas. University of New Mexico Press, Albuquerque.

Bent, A.C. 1942. Life histories of North American flycatchers, larks, swallows, and their allies. U.S. National Museum Bulletin 179.

Carothers, S.W., 1974. Breeding ecology and time-energy budget of male vermilion flycatchers and comments on the social organization of southwestern riparian birds. Ph.D. Dissertation, University of Illinois, Chicago.

Carter, F. 1937. Bird life at Twenty-nine Palms. Condor 39:210-219.

Fitzpatrick, J. 1980. Foraging behavior of neotropical tyrant flycatchers. Condor 82:43-57.

Friedmann, H. 1963. Host relations of the parasitic cowbirds. U.S. National Museum Bulletin 233.

Friedmann, H., and L.F. Kiff. 1985. The parasitic cowbirds and their hosts. Proceedings of the Western Foundation of Vertebrate Zoology 2.

Friedmann, H., L.F. Kiff, and S.I. Rothstein. 1977. A further contribution to knowledge of the host relations of the parasitic cowbirds. Smithsonian Contributions to Zoology 235.

Garrett, K., and J. Dunn. 1981. Birds of Southern California: Status and Distribution. Los Angeles Audubon Society, Los Angeles, California.

Grinnell, J. 1914. An account of the mammals and birds of the Lower Colorado Valley, with especial reference to the distributional problems presented. University of California Publications in Zoology 12:51-294.

Grinnell, J., and A.H. Miller. 1944. The distribution of the birds of California. Pacific Coast Avifauna 27.

Hubbard, J.P. 1978. Revised check-list of the birds of New Mexico. New Mexico Ornithological Society, Publication No. 6.

Hunter, W.C. 1984. Status of nine bird species of special concern along the Colorado River. Nongame Wildlife Investigations, Wildlife Management Branch Administration Report 84-2. California Department of Fish & Game, Sacramento.

Monson, G., A.R. Phillips. 1981. Annotated checklist of the birds of Arizona. University of Arizona Press, Tucson.

Patten, M.A. 1995. Checklist of the birds of Morongo Valley. Bureau of Land Management, Morongo Valley, California.

Patten, M.A., G. McCaskie, and P. Unitt. 2003. Birds of the Salton Sea: Status, Biogeography, and Ecology. University of California Press, Berkeley.

Patten, M.A. 2006. Vermilion flycatcher (*Pyrocephalus rubinus*) Department of Biology, University of California, Riverside, California.

Phillips, A.R., J.T. Marshall Jr., and G. Monson. 1964. The birds of Arizona. University of Arizona Press, Tucson.

Rea, A.M. 1983. Once a River. University of Arizona Press, Tucson, Arizona.

Root, T. 1988. Atlas of wintering North American birds: An analysis of Christmas Bird Count data. University of Chicago Press, Chicago, Illinois.

Rosenberg, K.V., R.D. Ohmart, W.C. Hunter, and B.W. Anderson. 1991. Birds of the Lower Colorado River Valley. University of Arizona Press, Tucson, Arizona.

Serena, M. 1981. The status of selected riparian birds along the lower Colorado River. Unpublished report to Nongame Wildlife Investigations, California Department of Fish and Game. 29 p.

Unitt, P. 2004. San Diego County bird atlas. Proceedings of the San Diego Society of Natural History 39.

Wolf, B.O., and S.L. Jones. 2000. Vermilion flycatcher (*Pyrocephalus rubinus*). *In:* A. Poole and F. Gill (editors), The Birds of North America, No. 484. The Birds of North America, Inc., Philadelphia, Pennsylvania.

ARIZONA BELL'S VIREO (*Vireo bellii arizonae*)

Legal Status

The subspecies Arizona Bell's vireo (*V. b. arizonae*) was proposed for Federal listing in 1981 as endangered because of dramatic population declines. The petition failed because significant populations of the subspecies existed in Arizona and New Mexico. California listed *arizonae* as endangered in 1988 (California 1988).

Distribution

Historic Range

The Arizona Bell's vireo, until the 1950s, was quite abundant and much more widespread. Grinnell (1914) called the Bell's vireo "one of the most characteristic avifaunal elements in the riparian strip." He estimated that one singing male occupied every 200 yards of willow habitat along the river's edge. Toward the middle of the century, Grinnell and Miller (1944) still classified this vireo as a common summer resident. By the early 1960s, *V. b. arizonae* was reported to be scarce everywhere along the lower Colorado River (LCR) (Monson 1960, Phillips et al. 1964). By 1980, it was estimated that no more than 50 pairs of vireos nested on the both sides of the LCR, south of Davis Dam (Edwards 1980).

Between 1976 and 1986, Rosenberg (1991) estimated that *V. b. arizonae* declined 57% on the LCR with most of the reduction occurring after a flood event in 1983. From 1974 to 1984, breeding *V. b. arizonae* occurred only in the vicinity of Needles, Topock Marsh, Bill Williams River National Wildlife Refuge, and Cibola National Wildlife Refuge, with a few scattered pairs near Parker and Poston, Arizona. A small population of *V. b. arizonae* nested near the tip of Nevada in 1975 and 1976.

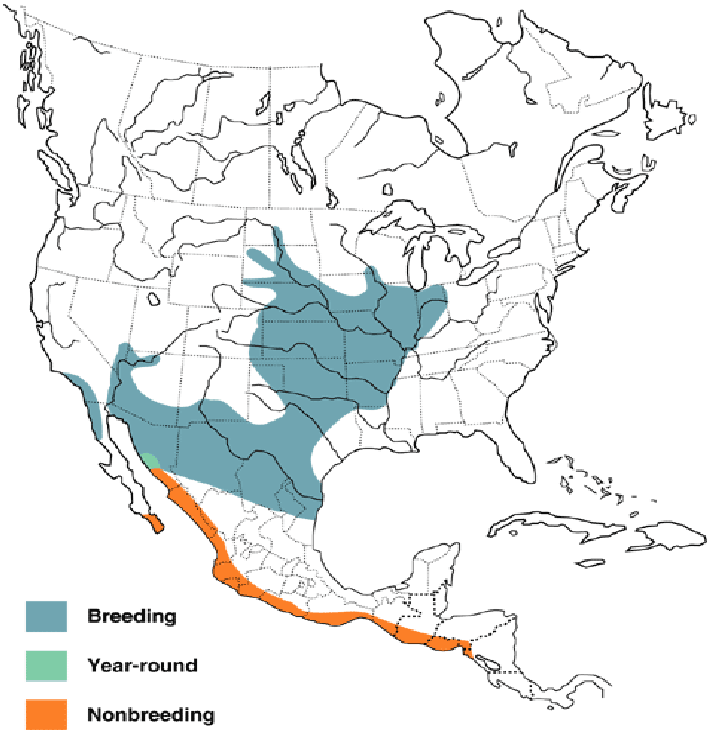
In 1981, 35 male *V. b. arizonae* were confirmed breeding in a study that focused on the California side of the LCR. Males were found in just two fairly restricted areas, from the Nevada border south to the beginning of Topock Gorge, and from just south of the Big River development (near Parker, Arizona) to 0.8 km south of Agnes-Wilson Bridge (Serena 1986). Laymon and Halterman (1986) found only four singing males in surveys of bird populations on the California side of the LCR and concluded that the Arizona Bell's vireo was on the verge of extirpation in California.

Current Range

The Bell's vireo is widespread in the central and southwestern United States and northern Mexico (see Figure 1). The Bell's vireo is known to breed in southern California, southern Nevada, southwest Utah, northwestern and southern Arizona, southern New Mexico, central and southwest Texas, eastern Colorado, central Nebraska, central South

Dakota, south-central North Dakota, southeast Minnesota, southern Wisconsin, northeast Illinois, and northwest Indiana, south to northern Baja California, southern Sonora, southern Durango, Zacatecas, southern Nuevo Leon, southern Tamaulipas, southern and eastern Texas, northwestern Louisiana, Arkansas, southwestern Tennessee, southwestern Kentucky, southern Indiana, and western Ohio (Brown 1993).

Figure 1. Bell's vireo species-wide breeding and wintering range. *V. b. arizonae* breed in southeastern California to southwestern New Mexico, and south to Sonora and Chihuahua, Mexico. The Arizona Bell's vireo is currently a rare to locally uncommon summer resident and breeder on the LCR (Rosenberg et al. 1991).



Populations within LCR MSCP Project Boundary

A remnant population of Arizona Bell's vireo persists on the Bill Williams River National Wildlife Refuge. In 1990, the Needles Municipal Golf Course supported patches of willow, mesquite trees, tall cottonwoods, and marshy ponds, which produced breeding Arizona Bell's vireos. Soto Ranch contained the last stand of mature honey mesquite on the California side of the LCR, and supported Arizona Bell's vireos locally (Rosenberg et al. 1991).

Riparian areas containing a mixture of willows and mesquites, north and south of Needles, California, have been known to support Arizona Bell's vireos (Serena 1986). Honey mesquite-saltcedar mixed stands represent the Arizona Bell's vireos most important habitat outside of the remaining willow habitats of the Bill Williams River National Wildlife Refuge and Needles, California (Rosenberg et al. 1991). A mixture of honey mesquite and saltcedar occurs locally on Cibola National Wildlife Refuge and on the Fort Mohave Indian Reservation. In this mixed plant community, saltcedar provides a dense understory, with honey mesquite offering accessible foraging sites for *V. b. arizonae* along with a well-developed, patchy canopy layer.

Life History

General description

The Bell's vireo is a small vireo, with a length of 115-125 mm and a weight of 7-10 g. This vireo has short, rounded wings, which makes the tail look long. The bill is short, straight, and slightly compressed at the base. Male and female Bell's vireos are sexually monomorphic in plumage color throughout the year. This plumage color varies from generally drab gray to green above, white to yellow below, with an unstreaked breast. The Bell's vireo has a faint white eye ring. There are two pale wing bars, with the lower bar more prominent. The plumage of juveniles resembles that of adults in worn summer plumage—essentially white and gray, but whiter below with more distinct wingbars (Brown 1993).

Breeding

The Arizona Bell's vireo is a summer resident that generally breeds between late March and late September. The earliest recorded arrival date for the LCR was 8 March and the latest departure date was from late November (Rosenberg et al. 1991).

According to Bent (1950), the Arizona Bell's vireo is a typical breeder of the streamside fringes of willows and mesquite along the lower Colorado River. Grinnell (1914) found this vireo at every station surveyed along the Colorado River and described them as one of the most characteristic birds of the riparian strip. They were closely confined to the willow association, with singing males occupying 200-yd segments of habitat. Each pair was closely restricted in foraging area by neighboring pairs, and they actively resented encroachment by others of their own species. Serena (1986) found that birds tended to clump their territories. Two to four males typically occupy territories in a 400-800 m stretch, with large stretches of identical unoccupied habitat between groups of territories. Serena hypothesized two possible explanations. First, young males may preferentially establish territories in the vicinity of where they were born. Second, important criteria of desirable habitat may be the presence of nearby singing males or nesting pairs.

Bent (1950) reported that birds apparently are mated on arrival and nest construction begins almost immediately. Construction usually lasts 4 to 5 days and is done by the female. The nest is usually placed less than 1.5 m above the ground, with 1 m being the average height. Bent found nests in the following locations: 1 m above the ground on a horizontal willow branch beneath a clump of small willows, attached to a forking stalk of a seep-willow about 1.2 m above the ground, hanging between forks of a mesquite branch about 1.2 m above the ground, and suspended between two twigs and close to the stem of a slender willow about 2.8 m above the ground. Bent described a nest as a typical vireo basket not too firmly attached to twigs and made of various vegetable fibers such as split large grasses, and mixed with strips of soft inner bark, fine grasses, willow cotton, plant down, spider nests, and considerable cattle hair, all firmly bound together. The lining typically is constructed of the very finest grass tops and a little cattle hair. The nest is about 7 by 6 cm in outside diameter and 4.5 cm in outside depth, and internally it is about 4.4 by 1.8 cm in diameter and 3 cm deep. Three or four eggs usually are laid and both parents share in incubation, which lasts about 14 days (Bent 1950).

Although egg laying typically begins in early April, nests with eggs have been documented as early as 25 March in Arizona. Arizona Breeding Bird Atlas data reveal a strong peak in nesting activity from late April to late May, with some early broods fledged by 21 April. Nesting occurs through the summer. Atlasers reported a nest with young as late as 8 August and adults feeding dependent fledglings through 19 September (Averill-Murray and Corman 2005). Both parents participate in the care of the young, brooding them and feeding them mostly smooth caterpillars. Although normally timid, shy, and retiring, both parents are aggressive defenders of eggs and young. Two broods generally are raised each season. Juveniles undergo a partial post-juvenile molt in July and August. There is no pre-alternate molt, but rather a complete basic molt takes place in late summer.

Arizona Bell's vireos are common victims of nest parasitism by brown-headed cowbirds (Bent 1950). A female cowbird lays an egg in the nest of an absent vireo, usually removing one of the owner’s eggs if any are present, and then leaves, expecting the host to incubate and hatch the egg and raise the nesting. This results in a reduction of nest success and reduced productivity for the host bird. Usually the parasite nestling is much larger than the host’s own nestlings, and the parasite nestling simply out-competes them. Serena (1986) found that five of nine Arizona Bell's vireo's nests located along the California side of the Colorado River were parasitized by cowbirds in 1981. However, early Bell’s vireo nests often escape parasitism if initiated well in advance of the onset of cowbird egg-laying in mid- to late April (Averill-Murray and Corman 2005).

Diet

The Arizona Bell’s vireo is a summer visiting insectivore on the LCR (Rosenberg et al. 1991). Bent (1950) described the Arizona Bell's vireo as almost entirely insectivorous, and food items predominantly include various bugs, caterpillars, beetles, and grasshoppers. In one food habit study conducted during summer, Arizona Bell's vireos' overall diet consisted of 34.4% Hemiptera (true bugs) and 18.5% Orthoptera (grasshoppers). However, during July, orthopterans increased to 34.9% of the diet. Other items in the summer diet included adult moths and butterflies and their eggs, ladybird beetles, weevils, leafbeetles, bees and wasps, spiders, snails, and occasionally, wild fruit.

Grinnell (1914) reported that Bell's vireos foraged in all vegetation associations but were most represented in the willow association, especially where there was an undergrowth of seepwillow *(Baccharis glutinosa).* Bent reported that they worked in a rather low foliage zone, from the ground to a height of 6 to 8 feet.

Habitat

Early accounts indicate that *V. b. arizonae* was most common in willow-dominated habitats, where it occupied the understory shrubs like seepwillow (*Baccharis glutinosa*). Remnant LCR populations breed primarily in tall screwbean or honey mesquite woodlands near water, usually mixed with scattered willows and saltcedar. Arizona Bell’s vireos require a well-developed shrub layer, a dense understory, and at least a moderately tall canopy layer of vegetation for breeding and foraging (Rosenberg et al. 1991). *V. b. arizonae* also breeds in large stands of recently regenerated willows mixed with screwbean mesquite (Serena 1986). Serena (1986) found Goodding’s willow was the most important plant around nest sites in 18 of 35 *V. b. arizonae* breeding territories on the California side of the LCR. In all *V. b. arizonae* territories measured, willows occurred in small patches and were interspersed with other plants.

Bell’s vireos are habitat generalists east of the LCR, with higher densities in honey mesquite and saltcedar than in cottonwood-willow habitats at higher elevations (Hunter 1988). Bell’s vireos prefer to forage and breed in dense, low, shrubby vegetation in early successional stages in riparian areas, brushy fields, young second-growth woodland, scrub oak, coastal chaparral, and mesquite brushlands. Bell’s vireos are often near water in arid regions. Vireos nest in any successional stage with dense understory vegetation, with the most critical structural component of least Bell’s vireo habitat in California consisting of a dense shrub layer 0.6-3.0 m above ground (Goldwasser 1981, Franzreb 1989).

A procedure for developing a specific vegetation restoration model for the least Bell's vireo (*V. b. pusillus*) habitat utilized mean percent cover, density, abundance, species composition, and expected plant mortality rates (Baird and Rieger 1989). In one study, all five restoration sites monitored supported nesting *V. b. pusillus* within 3-5 years, providing the first evidence that it is possible to create suitable nesting habitat for this subspecies in coastal California lowlands. Nests at restoration sites successfully fledged young, and have been no less productive than nests in natural habitats. The key components of the site restoration were water availability, structure of planted vegetation, and the site's proximity to natural habitat (Kus 1998). Translocation and captive breeding for release into areas within their historical range has been analyzed but not undertaken (Franzreb 1990).

Threats

The near elimination of the Arizona Bell's vireo as a common breeding resident on the LCR has been attributed to a combination of loss of preferred willow habitats and increased pressure from parasitism by brown-headed cowbirds concurrent with agricultural development (Rosenberg et al. 1991). The Arizona Bell's vireo is a frequent host to brown-headed cowbirds on the LCR (Serena 1986, Rosenberg et al. 1991, Averill 1996, Averill and Morrison 1998).

Conservation Measures

BEVI1—Create 2,983 acres of Arizona Bell's vireo habitat. Of the 7,260 acres of created cottonwood-willow and honey mesquite, at least 2,983 acres will be designed and created to provide habitat for this species. Patches of created habitat will be designed and managed to support cottonwood-willow types III and IV and honey mesquite type III that provide habitat for this species. The created habitat will be established in patches as large as possible. In addition to the spatial replacement of affected habitat, the quality of created habitat will be substantially greater than affected habitats. Patches of existing cottonwood-willow in the LCR MSCP planning area typically include dense stands of saltcedar that support little vegetative diversity relative to the cottonwood-willow land cover that will be created as habitat. Created habitat will be dominated by native riparian trees (i.e., cottonwood and willow trees), support a tree structure corresponding to structural types III-IV, support a diversity of plant species, and will be created to the greatest extent practicable in patch sizes optimal for supporting the species. The design and management criteria described in the conservation measures for the southwestern willow flycatcher (Section 5.7.2) and yellow-billed cuckoo (Section 5.7.14) will ensure that created cottonwood-willow stands in structural types III and IV will also provide other habitat requirements for this species (e.g., habitat patch size, food requirements). In particular, the management of moist surface soil, slow-moving water, or ponded water conditions and greater diversity of seral stages of cottonwood-willow described in the conservation measures for the southwestern willow flycatcher habitat will also provide these habitat requirements for this species. Created habitat, thus, will approximate the condition of the native habitat of the species that was historically present along the LCR.

LCR MSCP Research and Monitoring Needs

Distribution, population status, demographics, habitat requirements, and threats need to be determined for Arizona Bell's vireo along the LCR. Distribution and population status will be determined through a multi-species system monitoring protocol. Habitat requirements, including vegetation composition, structure, microhabitat characteristics, patch size, density, and prey abundance need to be determined. A review is needed of habitat restoration for least Bell's vireo to gather information about site characteristics that can be applied to LCR restoration projects. The impact of brown-headed cowbird parasitism and predation needs to be evaluated.

Literature Cited

Averill, A. 1996. Brown-headed cowbird parasitism of neotropical migratory songbirds in riparian areas along the Lower Colorado River. M.S. Thesis. School of Renewable Natural Resources, University of Arizona.

Averill, A. and Morrison, M.L. 1998. Lower Colorado River Valley Cowbird Control Project. Annual Report. Final Draft, 28 March 1998. Submitted to the Lower Colorado River Region, Bureau of Reclamation.

Baird, K.J., and J.P. Rieger. 1989. A restoration design for least Bell's Vireo habitat in San Diego County. USDA Forest Service, Gen. Tech. Rep. PSW-110.

Bent, A.C. 1950. Life histories of North American wagtails, shrikes, vireos, and their allies. U.S. Natl. Mus. Bull. 197, U.S. Govt. Print. Office, Washington, D.C.

Brown, B.T. 1993. Bell’s Vireo. *In:* A. Poole, P. Stettenheim, and F. Gill (editors), The Birds of North America, No. 35. Philadelphia: The Academy of Natural Sciences. Washington, D.C.: The American Ornithologists’ Union.

Edwards, C.L. 1980. A report on the distribution, population trends and habitat trends and habitat requirements of the Bell’s vireo on the lower Colorado River. Prepared by Arizona Game and Fish Department for the Yuma district office of U.S. Bureau of Land Management.

Franzreb, K.E. 1989. Ecology and conservation of the endangered least Bell’s Vireo. Biol. Rep. 89, U.S. Fish and Wildl. Serv., Washington, D.C.

Franzreb, K.E. 1990. An analysis of options for reintroducing a migratory, native passerine, the endangered least Bell's vireo in the Central Valley, California. Biological Conservation 53(2):105-124.

Goldwasser, S. 1981. Habitat requirements of the least Bell’s Vireo. Final Rep., California Dept. of Fish and Game, Sacramento, California.

Grinnell, J. 1914. An account of the mammals and birds of the lower Colorado Valley. Univ. Calif. Pub. Zool. 12:51-294.

Grinnell, J., and A.H. Miller. 1944. The distribution of birds of California. Pacific Coast Avifauna 27. 615 p.

Hunter, W.C. 1988. Use of exotic saltcedar (*Tamarix chinensis*) by birds in arid riparian systems. Condor 90:113-123.

Kus, B.E. 1998. Use of restored riparian habitat by the endangered least Bell's Vireo. Restoration Ecology 6(1):75-82.

Monson, G. 1960. The nesting season. Southwest regional report. Audubon Field Notes 14:469.

Phillips, A.R., J.T. Marshall Jr., G. Monson. 1964. The birds of Arizona. University of Arizona Press, Tucson.

Rosenberg, K.V., R.D. Ohmart, W.C. Hunter, and B.W. Anderson. 1991. Birds of the Lower Colorado River Valley. University Of Arizona Press, Tucson.

Serena, M. 1986. Distribution, habitat preferences, and reproductive success of Arizona Bell's vireo (*Vireo bellii arizonae*) along the lower Colorado river in 1981. Final Report, California Department of Fish and Game, Sacramento.

SONORAN YELLOW WARBLER
(*Dendroica petechia sonorana*)

Legal Status

The Sonoran yellow warbler is not a Federally protected species. In California, this warbler is listed as a species of special concern (breeding), priority 2, and has not been included on prior special concern lists (Remsen 1978, CDFG 1992).

Distribution

Historical Range

Swarth (1914) characterized the Sonoran yellow warbler as a common summer resident confined almost entirely to the lower Sonoran river valleys, including the Colorado River Valley from Ft. Mohave to Yuma, Arizona. *Dendroica petechia sonorana* was formerly common along major rivers, like the Rio Grande and lower Colorado Rivers (Lowther et al. 1999). Given the great expanse of willow-cottonwood habitats at the turn of the nineteenth century, Rosenberg et al. (1991) speculated that the total size of the Sonoran yellow warbler population in the valley of the lower Colorado River was “enormous” at that time. In 1914, an estimated one to four males occurred in every 0.40 ha of willow and cottonwood habitat along the river (Grinnell 1914). In the 1940s, the Sonoran yellow warbler was an “abundant” breeder along the entire California side of the lower Colorado River Valley below 183 meters (Grinnell and Miller 1944). Since the 1930s, populations in California have declined throughout state along with the loss of breeding habitat, with steeper declines since the 1950s (Small 1994). Rosenberg et al. (1991) reported that the Sonoran yellow warbler bred “commonly” at the Bill Williams River and Topock Marsh in 1952 but had disappeared from these historic nesting sites after 1955. *D. p. sonorana* was considered extirpated from the valley by 1960 (Monson and Phillips 1981). In the late 1970s and early 1980s, singing males were reported sporadically along the California side of the LCR, and in 1986, one female was observed feeding a juvenile near Blythe (Garrett and Dunn 1981, Rosenberg et al. 1991). Since the 1960s, the Sonoran yellow warbler has been extirpated as a breeder from Texas north of 29°N, most likely due to the loss of riparian habitat (Oberholser 1974).

Current Range

Figure 1 represents the yellow warbler species-wide distribution. The current range of the Sonoran yellow warbler includes the southwestern portion of the United States into northern Mexico. By all known accounts, *D. p. sonorana* populations have fluctuated since their period of high abundance in the early 1900s. Despite population changes and local extirpations, the overall range of the Sonoran yellow warbler today has changed little since 1944 (Heath 2006). When considering historical cottonwood willow habitat on the LCR prior to the 1900s, *D. p. sonorana* numbers are likely far below historical population levels (Rosenberg et al. 1991). Reports of total extirpation along the lower

Colorado (e.g., Small 1994) possibly reflected localized extirpations rather than river-wide losses (McKernan and Braden 2002).

Winter Range

The yellow warbler is found wintering in small numbers in the United States. There are 8 records of wintering yellow warblers in northern California and 25 records of wintering birds in southern California, most from the LCR, Coachella, and Imperial valleys (Small 1994). There was one winter specimen collected from Topock Marsh from 7 February to 7 March 1951 (Phillips et al. 1964).

Populations within LCR MSCP Project Boundary

Sonoran yellow warblers were a common breeding bird on the LCR, but were considered extirpated as a breeding species throughout the lower Colorado River floodplain by the mid-1950s (Rosenberg et al. 1991). Recently, the species has been detected within the LCR watershed, with confirmed breeding pairs recorded in at least 4 of the 6 years from 1996 to 2001 at the following study sites: Virgin River, Pahranagat, Grand Canyon, Topock Marsh, Topock Gorge, Bill Williams, and Clear Lake. Breeding was confirmed for 2 years at these study sites: Meadow Valley, Lake Havasu, Headgate Dam, Hall Island, Cibola Lake, Walker Lake, Draper Lake, Paradise Valley, Adobe Lake, Clear Lake, and Taylor Lake (McKernan and Braden 2002). This species has also been found recently in areas being restored with native cottonwood and willow.

Figure 1. Distribution of yellow warbler (*aestiva* group), which includes the Sonoran yellow warbler. Individuals of this group also winter in northern South America (from Lowther et al. 1999).

Life History

General Description

The yellow warbler is a medium-sized, foliage-gleaning wood warbler (12-13 cm long, 9-11 g). Its plumage is more extensively yellow than most other wood warblers, and is unique in having yellow on the inner webs of the tail feathers, except for the middle pair. The yellow warbler has indistinct wing bars. Yellow warbler males exhibit rather distinct geographic variation both within and among the three taxonomic groups. The variation of female and immature yellow warblers is also wide ranging but less well known. The yellow warbler is a widespread species in North America, breeding as far north as the tundra regions of Canada (Lowther et al. 1999). The yellow warbler remains common in much of its range as a habitat generalist.

Recent taxonomic review listed 43 recognizable subspecies, arranged into three groups mainly based on the adult male head color: 1) yellow warbler (*aestiva* group), yellow-headed, migratory forms breeding in North America, 2) golden warbler (*petechia* group), largely chestnut-capped, resident forms in the West Indies, and (3) mangrove warbler (*erithachorides* group), chestnut-hooded, resident forms of coastal middle and northern South America (Browning 1994).

This species profile focuses on the subspecies that breeds in the southwestern United States and northern Mexico. *Dendroica petechia sonorana,* the Sonoran yellow warbler, breeds only along the lower Colorado River in California, and from southern Arizona and southwest New Mexico to northeast Baja California Norte, interior Nayarit and Lacatecas, Mexico, and possibly the Colorado River Delta (Browning 1994, McKernan and Braden 2002, Lowther et al. 1999).

The *aestiva* group, male yellow warbler in breeding plumage has a bright yellow face, throat, and underparts, and is variably streaked with chestnut below the throat. The upperparts are yellow-green to olive with wing feathers edged yellow. The dark eye stands out on a comparatively unmarked yellow face. The male *aestiva* appears rather short-tailed and has yellow tail spots. The *aestiva* breeding female is similar to the male but less boldly marked, dull green on the upperparts, and has reduced chestnut streaking on the underparts. The plumage generally lacks distinctive markings, except for ventral streaking. The face pattern is plain, except for an indistinct yellowish eye ring. Adults in non-breeding plumage are similar, but duller and more greenish above. The streaking on the underparts is somewhat obscured by the yellowish feather tips. Hatch-year *aestiva* yellow warblers are duller than adults of the same sex, are more greenish, with streaking on underparts reduced or lacking, and have a whitish or pale yellowish eye ring (Lowther et al. 1999).

Breeding

Historically, the Sonoran yellow warbler bred in the willow and cottonwood habitats that lined the LCR. In surveys conducted from 1996 to 2002, more 75% of 100+ *D. p. sonorana* nests found downstream of Davis Dam on the lower Colorado River were in tamarisk (R. McKernan pers. comm. from Heath 2006). Basic life history traits of *D. p.*

sonorana, such as number of nesting attempts and extent of double brooding adult or juvenile survivorship rates, are unreported.

In Arizona, yellow warblers begin arriving on their breeding grounds in April, with the earliest arrival reported on 1 April. Atlasers observed nest building activity by 16 April. Egg laying likely begins in late April based on observations of nests, with young by 9 May. Active yellow warbler nests were reported from 28 April to 14 July. An adult was observed feeding a fledgling as late as 10 August (Wise-Gervais 2005).

Diet

There are no specific studies on the diet of *sonorana*. The yellow warbler, however, is a generalist species that appears to adapt its foraging to variation in local vegetation structure (Petit et al. 1990). Yellow warbler diet in California contained more than 97% animal matter, including ants, bees, wasps, caterpillars, beetles, true bugs, flies, and spiders (Beal 1907).

Habitat

Little adequate data currently exists to determine habitat use by *sonorana.* In general, yellow warblers are closely associated with moisture-loving deciduous trees throughout much of their extensive North American range. In the arid West, this preference leads them primarily to cottonwood and willow dominated riparian areas. In Arizona, yellow warblers were reported in Fremont cottonwood-willow associations, with a dense understory of deciduous saplings, seepwillow, mesquite, and tamarisk (Wise-Gervais 2005).

Yellow warblers make use of saltcedar and Athel tamarisk as both a nest substrate plant and as nesting habitat along the Colorado River in Grand Canyon and upper Lake Mead, where they have been identified as habitat generalists (Brown and Trosset 1989). Transient yellow warblers along the LCR make use of dense riparian vegetation, including saltcedar, and Athel tamarisk. Wintering warblers appear most common in planted trees around trailer parks, such as near Parker, Earp, and Lost Lake (Rosenberg et al. 1991).

Yellow warblers were quick to respond to habitat recovery after the removal of cattle from breeding areas (Taylor and Littlefield 1986, Kreuper et al. 2003). Sonoran yellow warblers constructed nests in willow stands and revegetated cottonwoods at Lake Havasu, Arizona (Lynn and Averill 1996).

Threats

An estimated 160,000 to 180,000 ha of native riparian vegetation was estimated to have been along the LCR in 1894 (Mearns 1907). By 1986, this native riparian vegetation had been reduced to roughly 25% (40,000 ha) of its former extent (Anderson and Ohmart 1984, Younker and Anderson 1986). The halting of annual flooding, agricultural, and urban development within historic floodplains, tamarisk invasions, and the death of much of the remaining riparian vegetation from excessive flood control releases has changed the structure, plant species composition, and function of the lower Colorado River's riparian system. Rosenberg et al. (1991) suggested that the resulting major losses of willow-cottonwood riparian on the river were the initial and primary cause of yellow warbler declines.

Rosenberg et al. (1991) speculated that Sonoran yellow warblers nesting in replacement habitats of screwbean mesquite (*Prosopis pubescens*) and tamarisk may have experienced higher rates of breeding failure than in native habitats, thereby causing further declines of the species.

Cowbird parasitism poses a limited to moderate threat to yellow warblers. However, the current impact of cowbird parasitism remains unreported for *D. p. sonorana.* East of the Sierra Nevada crest, 41% of 566 *D. p. morcomi* nests were parasitized. Yellow warbler young fledged from 37% of parasitized nests, and predation accounted for 55% of nest loss in parasitized nests. Demonstrating a somewhat different response, yellow warblers nesting in tamarisk at Amargosa Canyon, Inyo County, fledged from only 2 of 16 parasitized nests (23 total), but predation rates of parasitized nests were similar to those found in the eastern Sierra (Heath 2006).

The effects of predation on *D. p. sonorana* productivity and population viability are unreported.

Conservation Measures

YWAR1—Create 4,050 acres of Sonoran yellow warbler habitat.
Of the 5,940 acres of created cottonwood-willow, at least 4,050 acres will be designed and created to provide habitat for this species. Patches of created habitat will be designed and managed to support cottonwood-willow types I-IV. The created habitat will be established in patches as large as possible. At a minimum, however, all of the habitat will be created in patches of at least 10 acres, and thus, based on the best available information, will meet the minimum habitat patch size requirements of the species. Created riparian forests will support breeding and migration habitats for yellow warblers that migrate along the LCR. In addition, the per-acre quality of created habitat for this species will be substantially greater than that of the affected habitat.

Along the LCR, this species formerly nested in cottonwood-willow habitat ranging from gallery forests to early successional stage scrublands. Patches of existing cottonwood-willow in the LCR MSCP planning area typically include dense stands of saltcedar that support little vegetative diversity relative to the cottonwood-willow land cover that will be created as habitat. Created habitat will be dominated by native riparian trees (i.e., cottonwood and willow trees), support a tree structure corresponding to structural types I-IV, support a diversity of plant species, and be created to the greatest extent practicable in patch sizes optimal for supporting the species. Created habitat, thus, will approximate the condition of the native habitat of the species that was historically present along the LCR. The design and management criteria described in the conservation measures for the southwestern willow flycatcher (Section 5.7.2) and yellow-billed cuckoo

(Section 5.7.14) will ensure that created cottonwood-willow stands in structural types I-IV will also provide other habitat requirements for this species (e.g., habitat patch size, food requirements).

LCR MSCP Research and Monitoring Needs

Research and monitoring needs include: 1) determining current population, distribution, and trends for Sonoran yellow warbler along the LCR, and factors limiting distribution and abundance, 2) conducting multi-species system monitoring to determine distribution and population trends for other avian species, 3) determining habitat characteristics necessary for breeding populations of *D. p. sonorana*, including vegetation composition, structure, density, microhabitat requirements, and prey availability/abundance, and 4) determining brown-headed cowbird parasitism rates and impacts of parasitism and predation on nest survivorship and fecundity of Sonoran yellow warblers.

Literature Cited

Anderson, B.W., and R.D. Ohmart. 1984. A vegetation management study for the enhancement of wildlife along the lower Colorado River. Final report to Bureau of Reclamation, Boulder City, NV.

Beal, F.E.L. 1907. Birds of California in relation to the fruit industry, part 1. U.S. Department of Agriculture Biological Survey Bulletin 30.

Brown, B.T., and M.W. Trosset. 1989. Nesting-habitat relationships of riparian birds along the Colorado River in Grand Canyon, Arizona. The Southwestern Naturalist 34:260-270.

Browning, M.R. 1994. A taxonomic review of *Dendroica petechia* (yellow warbler). Proceedings of the Biological Society of Washington 107:27-414.

California Department of Fish and Game. 1992. Bird species of special concern. Unpublished list, July 1992, California Department of Fish and Game, Sacramento, California.

Garrett, K., and Dunn, J. 1981. Birds of Southern California: Status and Distribution. Los Angeles Audubon Society, Los Angeles.

Grinnell, J. 1914. An account of the mammals and birds of the lower Colorado Valley with especial reference to the distributional problems presented. University of California Publications in Zoology 12:51-294.

Grinnell, J., and A.H. Miller. 1944. The distribution of the birds of California. Pacific Coast Avifauna 27.

Heath, S.A. 2006. Sonora yellow warbler (*Dendroica petechia sonorana*) Habitat Conservation Planning Branch. Available at http://www.dfg.ca.gov/hcpb/bsscrevu/bsscindex.shtml.

Hunter, W.C. 1984. Status of nine bird species of special concern along the Colorado River. Nongame Wildlife Investigations, Wildlife Management Branch Administrative Report 84-2. California Department of Fish and Game, Sacramento.

Krueper, D., J. Barth, and T.D. Rich. 2003. Response of breeding birds to the removal of cattle on the San Pedro River, Arizona. Conservation Biology 17:607-615.

Lowther, P.E., C. Celada, N.K. Klein, C.C. Rimmer, and D.A. Spector. 1999. *In:* A. Poole, (editor), Yellow Warbler (*Dendroica petechia*). The Birds of North America Online. Available at http://bna.birds.cornell.edu/BNA/account/Yellow_Warbler/.

Lynn, S., and Averill, A. 1996. Neotropical migratory bird monitoring project in the lower Colorado River Valley, final report, 1994-1995. Submitted to U.S. Fish and Wildlife Service, Region 2, Albuquerque, New Mexico.

McKernan, R.L., and Braden, G. 2002. Status, distribution, and habitat affinities of the southwestern willow flycatcher along the lower Colorado River – year 6, 2001. Biological Sciences Division, San Bernardino County Museum. Redlands, California.

Mearns, E.A. 1907. Mammals of the Mexican border of the United States. A descriptive catalogue of the species of mammals occurring in that region; with a general summary of the natural history, and a list of trees. Smithsonian Institution, U.S. Natural Museum Bulletin 56.

Oberholser, H.C. 1974. The bird life of Texas. University of Texas Press, Austin.

Ohmart, R.D., B.W. Anderson, and W.C. Hunter. 1988. The ecology of the lower Colorado River from Davis Dam to the Mexico-United States International Boundary: A community profile. U.S. Fish and Wildlife Service Biological Report 85 (7.19).

Petit, D.R., K.E. Petit, and L.J. Petit. 1990. Geographic variation in foraging ecology of North American insectivorous birds. Studies in Avian Biology 13.

Phillips, A.R., J. Marshall, and G. Monson. 1964. The Birds of Arizona. University of Arizona Press, Tucson.

Remsen, J.V. Jr. 1978. Bird species of special concern in California: An annotated list of declining or vulnerable bird species. Nongame Wildlife Investigations, Wildlife Management Branch Administrative Report 18-1. California Department of Fish and Game, Sacramento, California.

Rosenberg, K.V., R.D. Ohmart, W.C. Hunter, and B.W. Anderson. 1991. Birds of the lower Colorado River Valley. University of Arizona Press, Tucson.

Sauer, J.R., J.E. Hines, and J. Fallon. 2005. The North American Breeding Bird Survey, results and analysis 1966-2004, version 2005.2. USGS Patuxent Wildlife Research Center, Laurel, Maryland. Available at http://www.mbr-pwrc.usgs.gov/bbs/bbs.html.

Small, A. 1994. California Birds: Their Status and Distribution. Ibis Publishing Co., Vista, California.

Smith, J.N.M. 1999. The basis for cowbird management: Host selection, impacts on hosts, and criteria for taking management action. Studies in Avian Biology 18:104-108.

Studd, M.V., and R.J. Robertson. 1989. Influence of age and territory quality on the reproductive behavior of male yellow warblers. Canadian Journal of Zoology 67:268-273.

Swarth, H.S. 1914. A distributional list of the birds of Arizona. Pacific Coast Avifauna 10:1-133.

Taylor, D.M., and C.D. Littlefield. 1986. Willow flycatcher and yellow warbler response to cattle grazing. American Birds 40:1169-1173.

Wise-Gervais, C. 2005. Yellow warbler *Dendroica petechia. In:* Corman, T. E. and Wise-Gervais (editors), The Arizona Breeding Bird Atlas. University of New Mexico Press, Albuquerque.

Younker, G.L., and C.W. Anderson. 1986. Mapping methods and vegetation changes along the lower Colorado River between Davis Dam and the border with Mexico. Final report to Bureau of Reclamation, Lower Colorado Region, Boulder City, Nevada.

SUMMER TANAGER
(*Piranga rubra*)

Legal Status

The summer tanager is not currently Federally listed. In California, the tanager is considered a Bird Species of Special Concern (breeding), Priority 1, and has been included on the list since its inception (Remsen 1978, CDFG 1992). It is a covered species under the Clark County Multi-Species Habitat Conservation Plan (MSHCP).

Distribution

Historical Range

The summer tanager has been declining along edges of its range in most areas of eastern United States. The tanager formerly bred in central Iowa, southern Wisconsin, throughout northern Illinois, and central Indiana (Robinson 1996). In the West, the tanager was formerly considered to be common in the lower Colorado River Valley by Grinnell (1914), but only 216 individuals were estimated to be present there by 1976 (Rosenberg et al. 1991). Habitat destruction is the likely cause of population decline in the Lower Colorado River Valley (Hunter 1984); causes of decline along the northern edge of the eastern range are unknown.

Grinnell and Miller (1944) described the summer tanager's breeding range in California as the lower Colorado River, from the Nevada line south to the Mexican border. They list specific records from Needles, California, north to the Nevada line, 25 miles below Ehrenberg, Arizona, and from 8 miles below Picacho State Recreation Area in California, downstream to Pilot Knob. Cooper (1861), the earliest ornithologist to visit the lower Colorado River Valley, found the summer tanager to be "common," as did Grinnell (1914). Grinnell and Miller (1944) considered it to be "common within restricted range and habitat."

Current Range

The summer tanager breeds across the southern United States and northern Mexico, and winters from central mainland Mexico south to northern South America (Robinson 1996, AOU 1998). Two subspecies are currently recognized. One, *P. r. cooperi*, breeds from southern California east to New Mexico and south in mainland Mexico to the states of Durango and Nuevo Leon. The other, *P. r. rubra*, breeds from central-west Texas across the Gulf coast states to Florida and north to about 40°N (AOU 1957, Robinson 1996). Because of its attachment to mature riparian forest, this tanager is localized over its western range, although "common" within this habitat in Arizona (Monson and Phillips 1981). Densities can reach 20-30 birds/ha in cottonwood-willow stands along the Lower Colorado River (Rosenberg et al. 1991).

By 1976, numbers had declined tremendously, with only 216 individuals estimated along the lower Colorado River, well over half of these on the Arizona side, primarily in the Bill Williams River delta (Rosenberg et al. 1991). The population continued to dwindle to an estimated 198 individuals in 1984 and 138 in 1986, reflecting habitat losses from flooding (Rosenberg et al. 1991). In 1983, Hunter (1984) estimated a population of 46 summer tanagers for the California side of the LCR on the basis of available habitat. However, survey results caused Hunter to decrease the estimated population to no more than 10 pairs on the California side of the river. The most recent comprehensive survey, conducted in 1986, detected 22 males, only 3 on the California side of the river (S.A. Laymon and M. Halterman in Rosenberg et al. 1991).

By contrast to this precipitous decrease along the Colorado River, the summer tanager elsewhere in southern California has gradually colonized and spread. The species was first reported from Morongo Valley in 1962 and from the South Fork of the Kern River in 1977. Some breeding groups, consisting of as few as a single pair, have been irregular or ephemeral; others, most notably the breeding group on the South Fork of the Kern River, have increased impressively. Many sites have not been surveyed regularly, and therefore, summer tanager populations cannot be ascertained (Unitt 2006). Along the South Fork of the Kern River near Weldon, Kern County, the population stabilized at 30-38 pairs from 1985 through 1995, then rose to 35-45 pairs from 1994 through 2000 (Robinson 1996). The total known California population of summer tanagers is just over 100 pairs (Unitt 2006).

Figure 1. Distribution of the summer tanager in North and Middle America. This species winters south to northern South America (Robinson 1996).

Populations within LCR MSCP Project Boundary

McKernan and Braden (2002) summarized summer tanager observations from 1996 to 2001. Breeding tanagers were confirmed for at least 2 years at the following sites: Virgin River, Pahranagat, Meadow Valley, Grand Canyon, Topock Marsh, Topock Gorge, Lake Havasu, Bill Williams, Headgate Dam, Ehrenberg, Walker Lake, Draper Lake, Paradise Valley, Adobe Lake, and Taylor Lake.

Eight to 10 pairs are estimated to occur in the 69-ha Athel tamarisk stand near Topock, Arizona (Rosenberg et al. 1991). In Picacho State Recreation Area, breeding tanagers were observed feeding at least one fledgling on 19 July 2000. Between 1999 and 2001, summer tanagers were observed irregularly in an 8-ha revegetation site adjacent to the main campground at Picacho State Recreation Area (Unitt 2006).

Life History

General Description

The summer tanager is a large tanager, about 17 cm long, with an average mass of nearly 30 g. The summer tanager is most easily confused with the congeneric scarlet (*Piranga olivacea*) and hepatic (*P. flava*) tanagers. Adult males are distinguished from the scarlet tanager by paler plumage, with more rose or orange-red than intense scarlet, and red, rather than black, wings and tail. Adult male hepatic tanagers have dusky gray ear patches, and even duller red plumage that often appears gray. Female summer tanagers are usually brownish or orange-yellow, lack greenish cast to plumage, and have narrow, yet conspicuous, yellowish edging on wing coverts that make the wings appear to have the same color as the body. Some older female summer tanagers apparently become partly or even completely pigmented as in males, with plumage ranging from yellow to orange-red to red. Some females even show a patchy plumage with several shades of red scattered across body. Typical immature male summer tanagers have a distinctive spotted or splotched plumage with orange-red and red patches on an otherwise yellowish plumage. Many immatures may be indistinguishable from females. Summer tanagers are easily distinguished from the western tanager (*P. ludoviciana*) by a lack of white wing-bars. The summer tanager call is a distinctive pit-i-tuck, more staccato than that of the western tanager and very different from the calls of the scarlet (chip-brrr) and hepatic (soft chuk) tanagers (Robinson 1996).

Breeding

In Arizona, summer tanagers typically begin to arrive on their breeding grounds in mid-April to early May, with the earliest recorded arrival on 7 April. Males arrive first and aggressively establish territories by the end of April and beginning of May. Nest building has been recorded as early as 27 April, with egg laying occurring by mid-May. The earliest recorded nests with young occurred on 16 May. Peak nesting activity occurs from mid-May to early July, continuing well into August. First broods fledge in mid- or late June, with most pairs re-nesting with second broods in late July. An adult was observed feeding a fledgling as late as 11 September (Corman 2005). The summer tanager is a rare to uncommon summer breeder along the LCR.

Diet

Summer tanagers forage primarily for large insects as they move deliberately through the canopy of tall riparian trees, sallying for aerial prey or snatching insects from the foliage or branches while in flight (Rosenberg et al. 1991). The midsummer diet (n = 7 stomachs) on the lower Colorado River was mainly cicadas, bees and wasps, and grasshoppers, with a few spiders, beetles, flies, and bugs. During the late breeding season, migration, and winter, summer tanagers also consume fruit (Robinson 1996).

Habitat

The summer tanager prefers structurally well-developed cottonwood-willow stands where they attain densities of 20-30 birds per 40 ha (Rosenberg et al. 1991). In Arizona, summer tanagers have bred in stands of exotic Athel tamarisk (*Tamarix aphylla*), and at higher elevations, honey mesquite (*Prosopis glandulosa*) and tamarisk (*Tamarix* spp.). Rosenberg et al. (1991) suggested that tree height (at least 9 m) and canopy closure are the critical variables making habitat suitable for summer tanagers. They noted the species nests in tamarisk and mesquite at higher elevations farther east in Arizona, where the cooler temperatures mean that the shading qualities of the willows and cottonwoods are less critical to successful nesting. Summer tanagers are found mostly in two riparian communities types in Arizona. They attain their highest densities along perennial drainages where continuous woodlands of large Fremont cottonwood and Goodding's willow exist. Clearly, tall, shady trees are the most critical element (Corman 2005).

Along the South Fork of the Kern River, Gallion (in Robinson 1996) found summer tanagers using areas of 9 to 11 ha. Along the Colorado River, Rosenberg et al. (1991) recorded a density of 20-30 birds per 40 ha of suitable habitat.

Threats

Removal of riparian forest is the most direct threat to the summer tanager in California. In addition, habitat degradation, through fragmentation and the lowering of water tables, compound the effects of clearing, cutting, and burning of trees. Assuming the heat-moderating qualities of leafy cottonwoods and willows are critical to the nesting success of the summer tanager and other desert birds nesting in midsummer, as implied by Rosenberg et al. (1991), then fragmentation of a once-continuous forest could result in a lack of necessary cooler microclimates. Temperatures, even in the shade of remaining scattered cottonwoods, could rise above the critical threshold, killing some eggs or chicks. Patch size may be critical; Rosenberg et al. (1991) found that the birds failed to recolonize a 30-ha revegetation site grown to mature cottonwoods, although they visited it occasionally.

Unnatural water regimes, in combination with the invasion of tamarisk, are also a threat. Floods in 1983, 1984, and 1986 killed most remaining cottonwoods along the Lower Colorado River, and high soil salinity, prolonged inundation, and fire favored their replacement by tamarisk (Rosenberg et al. 1991). There has been virtually no regeneration of cottonwoods in the Imperial Valley for the past 25 years, and the number of trees is now decreasing rapidly as old trees die off. In addition to tamarisk, proliferation of other exotic plants, such as giant reed (*Arundo donax*) and Russian olive

(*Elaeagnus angustifolius*), displaces suitable summer tanager habitat. The spread of *Arundo* (accelerated by flooding in 1993) on the coastal slope threatens habitat into which the summer tanager could spread.

Fire is a serious threat to summer tanager habitat. Burning of riparian forest along the Colorado River favors tamarisk at the expense of cottonwood (Rosenberg et al. 1991). In the desert, regeneration of native riparian forests following fire, even without competition from exotic plants, is likely slower than on the better-watered coastal slope (Unitt 2006).

Cowbird parasitism has not been identified as a serious threat to the summer tanager in California, but the extent of this parasitism remains poorly studied, in part because the birds nest high in the canopy. Along the South Fork of the Kern River, only 1 of 16 nests was parasitized by the brown-headed cowbird (*Molothrus ater*) (T. Gallion in Robinson 1996). The brown-headed cowbird was “common” along the Colorado River in 1910 (Rosenberg et al. 1991), and it may pose a population-level threat only when tanager numbers are already greatly reduced by habitat loss and degradation. The larger bronzed cowbird (*Molothrus aeneus*) may pose more of a threat to the summer tanager than the smaller brown-headed cowbird. Bent (1958) called the summer tanager a “fairly regular” victim of the bronzed cowbird in Sonora, so the increase of the bronzed cowbird in California may raise concern (Unitt 2006).

Conservation Measures

SUTA1—Create 602 acres of summer tanager habitat.
Of the 5,940 acres of created cottonwood-willow, at least 602 acres will be designed and created to provide habitat for the species. Patches of created habitat will be designed and managed to support cottonwood-willow types I and II. The created habitat will be established in patches as large as possible. At a minimum, however, 4,050 acres of cottonwood-willow will be created in patches of at least 25 acres, and 1,890 acres will be created in patches of at least 10 acres.

In addition to the spatial replacement of affected habitat, the quality of created habitat will be substantially greater than affected habitats. Patches of existing cottonwood-willow in the LCR MSCP planning area typically include dense stands of saltcedar that support little vegetative diversity relative to the cottonwood-willow land cover that will be created as habitat. Created habitat will be dominated by native riparian trees (i.e., cottonwood and willow trees), support a tree structure corresponding to structural types I and II (i.e., over 50% of the trees are taller than 15 feet), support a diversity of plant species, and will be created to the greatest extent practicable in patch sizes optimal for supporting the species. Created habitat, thus, will approximate the condition of the native habitat of the species that was historically present along the LCR. The design and management criteria described in the conservation measures for the yellow-billed cuckoo (Section 5.7.14) will ensure that created cottonwood-willow stands in structural types I and II will also provide other habitat requirements for this species (e.g., habitat patch size, food requirements). In addition, created southwestern willow flycatcher habitat that supports cottonwood-willow types I and II could also provide habitat for this species.

LCR MSCP Research and Monitoring Needs

Current distribution and population trends for summer tanagers along the LCR have not been determined. The habitat requirements necessary to create breeding summer tanager habitat, especially minimum patch size, canopy closure, vegetation composition, structure, density, and microhabitat conditions are also currently not known.

Literature Cited

American Ornithologists' Union. 1998. Check-list of North American Birds, 7th ed. American Ornithologists' Union, Washington, D.C.

American Ornithologists' Union. 1957. Check-list of North American Birds, 5th ed. American Ornithologists' Union, Baltimore.

Bent, A.C. 1958. Life histories of North American blackbirds, orioles, tanagers, and allies. U.S. Natural Museum Bulletin 211.

California Department of Fish and Game. 1992. Bird species of special concern. Unpublished list, July 1992, California Department of Fish and Game, Sacramento, California.

Cardiff, E.A. 1993. Desert riparian-freshwater marsh [breeding bird census]. Journal of Field Ornithology 64 (suppl.):92-93.

Cooper, J.G. 1861. New California animals. Proceedings of the California Academy of Sciences 2:118-123.

Garrett, K., and J. Dunn. 1981. Birds of Southern California: Status and Distribution. Los Angeles Audubon Society, Los Angeles.

Corman, T.E. 2005. Summer Tanager. *In:* Corman, T. E. and Wise-Gervais (editors), The Arizona Breeding Bird Atlas. University of New Mexico Press, Albuquerque.

Grinnell, J. 1914. An account of the mammals and birds of the lower Colorado River with especial reference to the distributional problems presented. University of California Publications Zoology 12:51-294.

Grinnell, J., and A.H. Miller. 1944. The distribution of the birds of California. Pacific Coast Avifauna 27.

Hunter, W.C. 1984. Status of nine bird species of special concern along the Colorado River. Nongame Wildlife Investigations, Wildlife Management Branch Administrative Report 84-2, California Department of Fish and Game, Sacramento.

Johnson, N.K. 1994. Pioneering and natural expansion of breeding distributions in western North American birds. Studies in Avian Biology 15:27-44.

McKernan, R.L, and G. Braden. 2002. Status, distribution, and habitat affinities of the southwestern willow flycatcher along the lower Colorado River, Year 6—2001. Submitted to the Bureau of Reclamation, Lower Colorado River Region, Boulder City, Nevada.

Monson, G., and A.R. Phillips. 1981. Revised Checklist of Arizona Birds. University of Arizona Press, Tucson.

Myers, S.J. 1992. Biological inventory of the Mojave River corridor. *In:* Natural resources inventory of the Mojave River corridor, p. XX-XX. Report submitted to Mojave River Corridor Task Force by Tierra Madre Consultants. California Department of Fish and Game, Sacramento, California.

Myers, S.J. 1997. Summer Tanager (*Piranga rubra*). Account for the West Mojave Plan. Bureau of Land Management, Moreno Valley, California.

Phillips, A.R. 1966. Further systematic notes on Mexican birds. Bulletin of the British Ornithologists' Club 86:148–159.

Phillips, A., J. Marshall, and G. Monson. 1964. The Birds of Arizona. University of Arizona Press,Tucson.

Remsen, J.V. Jr. 1978. Bird species of special concern in California: An annotated list of declining or vulnerable bird species. Nongame Wildlife Investigations, Wildlife Management Branch Administrative Report 78-1. California Department of Fish and Game, Sacramento, California.

Robinson, W.D. 1996. Summer Tanager (*Piranga rubra*). *In:* A. Poole and F. Gill (editors), The Birds of North America, No. 248. Birds of North America, Philadelphia.

Rosenberg, K.V., R.D. Ohmart, W.C. Hunter, and B.W. Anderson. 1991. Birds of the Lower Colorado River Valley. University of Arizona Press, Tucson.

Sauer, J.R., J.E. Hines, and J. Fallon. 2005. The North American Breeding Bird Survey, results and analysis 1966-2004, version 2005.2. USGS Patuxent Wildlife Research Center, Laurel, Maryland. Available at http://www.mbr-pwrc.usgs.gov/bbs/bbs.html.

Shepherd, T. 2004. Analysis of intraspecific variation within the summer tanager (*Piranga rubra*) including a population-level study of *P. r. cooperi*. M.S. Thesis, San Diego State University, San Diego.

Unitt, P. 2004. San Diego County bird atlas. Proceedings of the San Diego Society of Natural History 39.

Unitt, P. 2006. Summer tanager (*Piranga rubra cooperi*). Habitat Conservation Planning Branch. Available at http://www.dfg.ca.gov/hcpb/bsscrevu/bsscindex.shtml.

FLAT-TAILED HORNED LIZARD (*Phrynosoma mcallii*)

Legal Status

The U.S. Fish and Wildlife Service published a proposed rule to list *Phrynosoma mcallii* as a threatened species on 29 November 1993. No critical habitat was determined at that time (USFWS 2005). The proposal to list *Phrynosoma mcallii* was withdrawn on 15 July 1997, for three primary reasons: 1) population trend data did not conclusively demonstrate significant population declines, 2) some threats to the occupied habitat of *Phrynosoma mcallii* had become less serious since the proposed rule was issued, and 3) the establishment of the 1997 Conservation Agreement and Rangewide Management Strategy (USFWS 2005). The 1993 proposal to list *Phrynosoma mcallii* was reinstated on 26 December 2001, withdrawn on 3 January 2003, reinstated on 7 December 2005, and withdrawn on 28 June 2006 (USFWS 2005 and 2006). The Bureau of Land Management (BLM) designated *Phrynosoma mcallii* as a sensitive species in 1980 (Wright 2003). The Mexican Government has designated *Phrynosoma mcallii* as a threatened species (FTHL ICC 2003).

Phrynosoma mcallii is listed as a species of special concern in the State of Arizona and a species of concern in the State of California (AFGD 2003). The California Fish and Game (CDFG) Commission designated *Phrynosoma mcallii* as a candidate species and recommended it be listed as a threatened species in the state of California on 13 May 1988 (Bolster and Nicol 1989). The Commission voted against the proposed listing on 22 June 1989 (Foreman 1997). Collecting for *Phrynosoma mcallii* is prohibited in California and Arizona (AFGD 2003). Natureserve ranks *Phrynosoma mcallii* as vulnerable to extirpation or extinction on a global level and imperiled on a national and state level in the states of Arizona and California (Natureserve 2005). Natureserve classifies vulnerable species as species that are at moderate risk of extinction due to a restricted range, relatively few populations (often 80 or fewer), recent and widespread declines, or other factors. Natureserve classifies imperiled species as those that are at high risk of extinction due to a restricted range, very few populations (often 20 or fewer), steep declines, or other factors (Natureserve 2005).

A Rangewide Conservation and Management Strategy, signed in 1997, formed a conservation agreement between signatory agencies (Foreman 1997). The purpose of this strategy was to secure and manage sufficient habitat to maintain several self-sustaining populations of *Phrynosoma mcallii* throughout the species range in the United States (Foreman 1997). The document was revised in 2003, and the implementation schedule is expected to be revised in 2008 (FTHL ICC 2003).

Distribution

Historical Range

Assessing the historical habitat of *Phrynosoma mcallii* was complicated by the fact that agriculture preceded knowledge of the species range in Imperial Valley, California, and Yuma Valley, Arizona (Hodges 1997). Hodges (1995) estimated that there were 203,520 acres of *Phrynosoma mcallii* historical habitat in Arizona. Piest and Knowles (2002) estimated that there were 221,043 acres of *Phrynosoma mcallii* historical habitat in Arizona. The northern and western boundary of *Phrynosoma mcallii's* historical range in Arizona is the Yuma Mesa. The eastern boundary is formed by the Fortuna Wash and Foothills boulevards, to just east of the sand dunes (Hodges 1997). Hodges (1997) estimated that there were 2.22 million acres of *Phrynosoma mcallii* historical habitat in California. The western boundary of *Phrynosoma mcallii's* historical range in California is formed by Fish Creek, Vallecito, and the Santa Rosa Mountains. The Yuha Basin habitat ends at the Sierra Juarez and Coyote mountains. A small valley of habitat stretches farther west along I-8 beyond Ocotillo and Coyote Wells, where I-8 joins Highway 92 and S2 forks north (Hodges 1997). Borrego Valley, between Vallecito and the Santa Rosa Mountains, contains *Phrynosoma mcallii* habitat, as does a valley between Indio Hills and the Little San Bernardino Mountains (Hodges 1997). The eastern extent of the range continues from East Mesa through the Algodones Dunes and is limited by new alluvial deposits from the Chocolate Mountains and Cargo Muchacho Mountains (Hodges 1997). The existence of *Phrynosoma mcallii* in Carrizo Valley, which is south of the Fish Creek Mountains and north of the Coyote Mountains, is unknown. The western limit and distribution in Borrego Valley is still relatively unknown (Hodges 1997).

Current Range

Phrynosoma mcallii occurs in the Sonoran Desert, extending from Coachella Valley (Riverside County) south to the head of the Gulf of California, taking in extreme southwest Arizona, northeast Baja California, and extreme northwest Sonora, Mexico (AFGD 2003; CDFG 1994; Rodrigues 2002). *Phrynosoma mcallii* has the smallest range of any horned lizard in the United States (Wright 2002).

Phrynosoma mcallii range in California extends from near the confluence of the San Gorgonio and Whitewaters Rivers in Riverside County, south and east through the Coachella Valley into Imperial County along both sides of the Salton Sea. From the area between the Salton Sea and San Diego County line, the range extends west into the Borrego Valley, although there may be important discontinuities in the badlands south of the Santa Rosa Mountains. *Phrynosoma mcallii* is generally restricted to elevations below 250 m in the Borrego Valley, and there are small extensions into the lower portions of the Coyote Creek Watershed, around Clark Dry Lake, and southwest along San Felipe Creek where it emerges from the Vallecito Mountains. *Phrynosoma mcallii* occurs east of the northeastern edge of the Callecitos and east and north of the Fish Creek Mountains, at increasingly lower elevations to below sea level in western Imperial County. *Phrynosoma mcallii* occurs east of Bow Willow in Carrizo Wash. It may occur in other flats or bajadas along Carrizo Wash and in drainages within the Carrizo Badlands. *Phrynosoma mcallii's* range extends eastward across East Mesa and the Algodones Dunes, except that the barren dunes are not inhabited. The Chocolate Mountains, Cargo Muchacha Mountains, and agricultural areas near Yuma, Arizona, probably separate California populations from those in Arizona. North of Niland, there may be a narrow band of habitat between the Salton Sea and the Chocolate Mountains continuous with the southeastern portion of the Coachella Valley (Natureserve 2005, Turner et al. 1980, Rorabaugh 1996B). *Phrynosoma mcallii's* range extends from the Borrego Badlands to Pilot Knob Mesa, east of the Algodones Dunes (Wright 2003). *Phrynosoma mcallii* occurs throughout much of the sandy flats and dune margins in the southern portion of the Salton Sea Test Base (Rorabaugh 1996 B and C). *Phrynosoma mcallii* inhabits 2,695 square miles in California; 330 square miles are considered optimal habitat (Rado 1981).

Phrynosoma mcallii range in southwestern Arizona extends southward of the Gila River and west of the Gila and Tinajas Atlas mountains in Yuma County (Townships T.9S, T.10S, T.11S, T.12S, Ranges R.24W, R.23W, R.22W, R.21W, R.20W) (Natureserve 2005; AFGD 2005; Rorabaugh et al. 1987). Hodges (1995) estimated that *Phrynosoma mcallii* inhabits 550-575 km^2 in Arizona. Rorabaugh et al. (1987) estimated that *Phrynosoma mcallii* inhabits 650-700 km^2 in Arizona. *Phrynosoma mcallii* range in Mexico extends southward from the Yuha Desert in California to Laguna Salada in Baja California, and southward from the Yuma Desert in Arizona, through the Pinacate region to the sandy plains around Puerto Penasco and Bahia de San Jorge, Sonora (Natureserve 2005, Rodrigues 2002). *Phrynosoma mcallii* does not occur contiguously across its range.

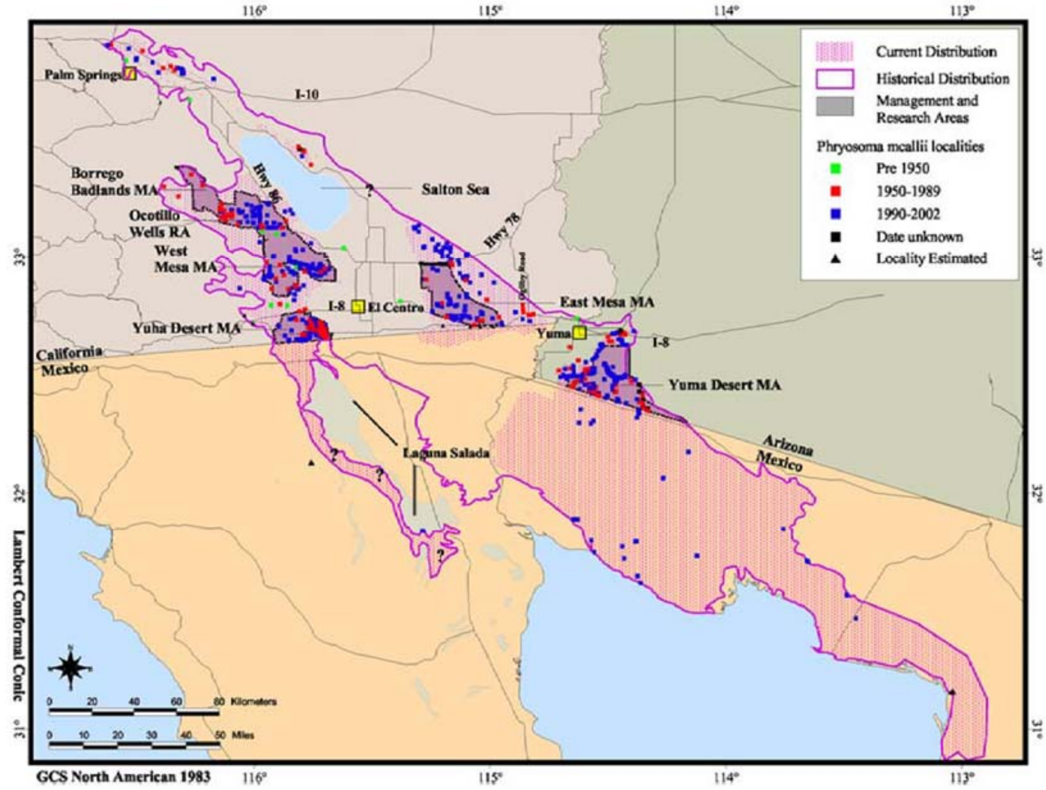
There are four geographically discrete populations in the United States (three in California and one in Arizona). The three in California are located in the Coachella Valley, the west side of the Salton Sea/Imperial Valley, and the east side of the Imperial Valley (Natureserve 2005). Populations in the Imperial Valley are divided into four major segments (Algodones Dunes, East Mesa, West Mesa/Anza Borrego, and Yuha) by Interstate 8 and the Coachella Canal (Wright 2002). Populations in the Coachella Valley are divided into two segments by Interstate 10 (Wright 2002). *Phrynosoma mcallii* and *Phrynosoma platyrhinos* are sympatric in portions of Arizona (T12S R 20/21W) and California (Hodges 1995, Rorabaugh et al. 1987).

Additional surveys are needed outside the management area to firmly delineate the boundaries on the exterior portion of *Phrynosoma mcallii's* range in the United States (Foreman 1997). Distribution of *Phrynosoma mcallii* in Mexico is poorly understood because few surveys in Mexico have been conducted (NatureServe 2005). Further studies need to be done in areas of Mexico, including surveys of: 1) the southeast boundary in Sonora, 2) the current range surrounding Mexicali Valley, 3) the current range surrounding the Laguna Salada, and 4) the degree of connectivity between the current ranges in Sonora, the Mexicali Valley, and surrounding Laguna Salada (Foreman 1997).

Populations within LCR MSCP Project Boundary

Phrynosoma mcallii occurs within reaches 6 and 7 of the Lower Colorado River Multi-Species Conservation Plan (LCR MSCP) program area (LCR MSCP HCP 2004). The Yuma Desert Management Area is the only *Phrynosoma mcallii* management area that lies within the LCR MSCP planning area (Foreman 1997).

Figure 1: *Phrynosoma mcallii* range in the United States and Mexico



Management and Research Areas

Five management areas and one research area were established for *Phrynosoma mcallii* in the 1997 Rangewide Management Strategy (Foreman 1997). Management areas include the majority of *Phrynosoma mcallii* habitat identified as key areas in previous studies; these areas minimize surface-disturbing and mortality causing activities (Foreman 1997). Management areas include land owned by the military, other Federal agencies, state agencies, and private land owners (Foreman 1997). The five management areas include the Yuma Desert Management Area (131,000 acres), the East Mesa Management Area (115,300 acres), the West Mesa Management Area (136,100 acres), the Yuha Basin Management Area (60,200 acres), and the Borrego Badlands Management Area (42,400 acres) (Foreman 1997). A research area at Ocotillo Wells State Vehicle Recreation Area (OWSVRA), encompassing 76,700 acres, is utilized for research on *Phrynosoma mcallii*. Research priorities focus on the effects of off road vehicles on *Phrynosoma mcallii* populations (Foreman 1997). Refer to the Flat-tailed Horned Lizard Rangewide Management Strategy for detailed information on boundaries of management and research areas (FTHL ICC 2003).

Current Abundance

Phrynosoma mcallii relative abundance indexes in Arizona are greatest (>15) in the four townships that are in the south central portion of the species' range (Rorabaugh et al. 1997). In California, *Phrynosoma mcallii* was the most abundant in four areas: 1) the southern part of East Mesa, particularly south of Ogilby in the vicinity of Gray’s Well, 2) the southeastern portion of the Yuha Basin and the vicinity of Signal Mountain, 3) south of Superstition Mountain, and 4) north of Ocotillo Wells and Benson Dry Lake (Turner et al. 1980). Relative abundance was determined using section searches.

Grant (2005) used closed mark-recapture analysis to estimate population size at three management areas from 2002 to 2004. Population estimates for the Yuha Basin Management Area in 2002 and 2004 were 25,514 lizards and 73,017 lizards, respectively (Grant 2005). Population estimates for the East Mesa Management Area and West Mesa Management Area in 2003 were 42,619 lizards and 10,849 lizards, respectively (Grant 2005). No overall trend can be inferred in the Yuha Basin Management Area from 2002 to 2004 as confidence intervals overlap (Grant 2005). The population estimate for the Yuma Desert Management area using mark and recapture analysis was 25,855 lizards (FTHL ICC 2003). The population estimate for the OWSVRA Research Area was 19,222 lizards (FTHL ICC 2003).

Life History

General Description

One of 14 species in the genus *Phrynosoma*, *Phrynosoma mcallii* was first collected by Colonel George A. M’Call and described by Hallowell in 1852 (Funk 1981, AGFD 2003). No subspecies of *Phrynosoma mcallii* has been described (Funk 1981). *Phrynosoma mcallii* is a moderate-sized, oviparous species of *Phrynosoma*, with an immaculate white venter, a narrow dark middorsal stripe from head to tail base, and a dorso-ventrally flattened tail (Funk 1981, AGFD 2003, CDFG 1994). The dorsal coloration is gray, tan, reddish-brown, or whitish (Funk 1981, CDFG 1994). There are two occipital spines 3-4 times longer than the basal width and not in contact at the base, and three temporal spines on each side of the body (Funk 1981, AGFD 2003). The nostrils lie within the canthal ridge. There is a single row of enlarged lateral gular scales, and the lateral abdominal fringe consists of two (occasionally three) rows of spines (Funk 1981). The ventral scales are smooth, and the peritoneum is black. The tympanum is not evident externally (Funk 1981, AGFD 2003). Males have enlarged postanal scales (Muth and Fisher 1992).

Adults weigh approximately 17 to 25 grams with snout to vent lengths (SVL) from 70 to 80 mm (Wright 2002). Males and females do not significantly differ in body size or growth rate (Muth and Fisher 1992). Hatchlings have SVLs of approximately 35-38 mm and weigh approximately 1.4 g (Bolster and Nicol 1989, Young and Young 2000). Gardner and Foley (2001) found SVL to be significantly correlated with weight and found appearance of seasonal deterioration in body condition.

Phrynosoma platyrhinos is the only other horned lizard known to be sympatric with *Phrynosoma mcallii.* Hybrids between the two species have been reported in Ocotillo, California and on the Barry M. Goldwater Range in Yuma, Arizona (Foreman 1997, AGFD 2003).

Breeding

Phrynosoma mcallii mates in April and May and first clutches are laid in May and June (AGFD 2003). Howard (1974) found that male *Phrynosoma mcallii* emerged from hibernation in April with testes and epididymides at maximum size. Spermatozoa were present in males until late July (Howard 1974). Howard observed that *Phrynosoma mcallii* eggs were present in May and hatchlings were present from July to October. *Phrynosoma mcallii* lay approximately 3 to 10 eggs per clutch and have up to two clutches per season (CDFG 1994, Muth and Fisher 1992). Average clutch size is 4.7 and there is a significant correlation between body size and clutch size of *Phrynosoma mcallii* (Howard 1974). *Phrynosoma mcallii* follows the reproduction strategy of multiple small clutches and early reproduction (Howard 1974). Howard (1974) found that of seven lizards in the *Phrynosoma* genera, *Phrynosoma mcallii* had the lowest productivity index. *Phrynosoma mcallii* reaches sexual maturity at age 1 year or less (Howland and Rorabaugh 1996). Howard (1974) found that the first clutch of individuals from July reached 54-58 mm by October and took part in reproduction their first season after hibernation. The second clutch of individuals that were 36-38 mm by October did not reach sexual maturity until their second season after hibernation (Howard 1974). There appears to be a pre-emergence yolk deposition in the majority of females (Howard 1974). Sex ratio of *Phrynosoma mcallii* is 1 male to 1 female (Turner et al. 1978).

Reproduction appears to be correlated with environmental conditions (Muth and Fisher 1992). Five centimeters of precipitation in the previous September to May is necessary for young of the year to reach breeding size by the next summer and for adults to be able to lay two clutches of eggs (Grant 2005). Reproduction may be at least doubled in wet years as opposed to dry years. Following heavy fall precipitation, hatchlings reached adult size in less than a year, while under drought conditions, it generally takes 2 years to reach breeding condition (Young and Young 2000). During years with heavy precipitation, laying and hatching can occur earlier, females can allocate more resources to producing more and/or larger eggs, and some females may lay multiple clutches in a year (Young and Young 2000). Results of the population viability analysis conducted by the flat-tailed horned lizard conservation team showed that variation in litter size affects population viability (Fisher et al. 1998).

Biology

There is no evidence of aestivation among *Phrynosoma mcallii* (Young and Young 2000). *Phrynosoma mcallii* are obligate hibernators that overwinter at 2.5 to 20 cm of depth in loose sand (CDFG 1994). The average depth of hibernation burrows in one population of *Phrynosoma mcallii* was 6.0 cm (Grant 2005). The entrance of hibernation burrows is plugged with substrate (Grant 2005). *Phrynosoma mcallii* hibernation behavior is variable (Grant 2005). Grant (2005) observed *Phrynosoma mcallii* entering hibernation burrows from early October to late December. Some individuals abandon their first hibernation burrows and dig new ones (Grant 2005). Body mass affects the date that lizards enter into hibernation. *Phrynosoma mcallii* delay hibernation to continue to grow or gain mass reserves for hibernation (Grant 2005). Several studies have estimated average winter dormancy for *Phrynosoma mcallii* at 85-89 days (Wone and Beauchamp 2003, Muth and Fisher 1992). Winter dormancy in *Phrynosoma mcallii* is controlled primarily by reduced photoperiod and reduced air temperature rather than reduced metabolic rate or body temperature (Mayhew 1965). *Phrynosoma mcallii* emerge from overwintering sites from late December through March when substrate temperatures at a depth of 5 cm reach their voluntary minimum temperature (CDFG 1994, Wone and Beauchamp 2003). Some juveniles remain active during the winter months (Grant 2005, NatureServe 2005).

Miller (1999) found that the area used by male and female *Phrynosoma mcallii* during the summer shifted through time and did not fit the definition of a home range. Miller (1999) found that males used an average of 3.55 ha and females used an average of 1.77 ha during the summer. Miller (1999) found that males moved significantly farther than females during a 15-day and 24-hour time period. Females that shift their area used significantly more area than males do (Miller 1999). Turner and Medica (1982) estimated the home range size for *Phrynosoma mcallii* males and females to be 0.12 ha and 0.05 ha, respectively. Muth and Fisher (1992) estimated the home range size for *Phrynosoma mcallii* males and females to be 1.78 ha and 1.97 ha, respectively. Wone and Beauchamp (2003) found that males had a significantly larger summer home range than females and were more active. Home range size may be dependent on the size of the individual, climatic conditions, or density of lizards (Wone and Beauchamp 2003, Young and Young 2000). Home ranges are not centered on obvious habitat features (Sester 2004). *Phrynosoma mcallii* show considerable overlap between home ranges, which may indicate lack of territorial behavior (Wone and Beauchamp 2003). *Phrynosoma mcallii* home range is significantly larger during breeding season than the non-breeding season (Wone and Beauchamp 2003).

Phrynosoma mcallii reaches its peak abundance from the months of April through October (Muth and Fisher 1992). Rainfall appears to be a factor in *Phrynosoma mcallii* abundance (Wright 1993, 2002; Wright and Grant 2003; Grant 2005). *Phrynosoma mcallii* populations can exhibit local boom and bust dynamics, and even local extinction and recolonization (Grant 2005). In drought conditions, growth and reproduction are limited and predation rates are higher, but population levels can increase rapidly after abundant fall and winter rainfall (Young and Young 2000).

Muth and Fisher (1992) observed that *Phrynosoma mcallii* spent 32% of the active period moving, 46% of the time they were motionless, 11% was spent feeding, and 11% was spent digging. *Phrynosoma mcallii* spend the night on the surface, fully exposed in the open (Young and Young 2000). *Phrynosoma mcallii* appear to partake in sand swimming for short distances, but swimming does not occur after the loss of momentum. *Phrynosoma mcallii* is capable of rapid locomotion but this is not sustained for long distances (Norris 1949). *Phrynosoma mcallii* avoids predators by diving in the sand in areas where aeolian sand is present. When sand is absent, *Phrynosoma mcallii* remain motionless or flee; when fleeing they run a short distance and stop, run into burrows, or run into a base of a shrub. When captured, *Phrynosoma mcallii* wiggle their head and dig their occipital horns in the handler’s hand, which may be an important escape behavior when captured by predators (Wone and Beauchamp 1995).

The optimal air temperature for *Phrynosoma mcallii* appears to be 35.2 to 40.2°C; when temperatures exceed 41°C individuals retreat underground (CDFG 1994, Wone and Beauchamp 2003, Wright 2002). *Phrynosoma mcallii* maintains optimal body temperature by orienting its body toward the sun or substrate (Norris 1949). The foraging strategy of *Phrynosoma mcallii* is intermediate to that of a sit-and-wait predator and active forager (Muth and Fisher 1992).

Diet

Phrynosoma mcallii feed on ants of the genera *Messor*, *Pogonomyrmex, Conomyrma*, and *Myrmecocystus* (AGFD 2003). They may also eat beetles and other arthropods (AGFD 2003). Turner et al. (1978) found that three species of harvester ants (*Veromessor pergandei, Pogonomyrmex californicus*, and *Pogonomyrmex magnacantha*) comprised 75% of all insects in *Phrynosoma mcallii* scat. A fourth species of ant, *Conomyrma insane*, comprised 16% of insects in the scat (Turner et al. 1978). Young and Young (2000) observed feeding rates of up to 80 harvester ants per 15 minutes. The number of ant colonies in an area may be an important habitat requirement for *Phrynosoma mcallii.* Several studies found the number of harvester ant colonies was correlated with high lizard abundance (Grant 2005, Rorabaugh et al. 1987, Turner and Medica 1982, Young 1998).

Phrynosoma mcallii primarily uses preformed water (water found in their food) to maintain proper water balance (AGFD 2005). Rain harvesting may provide an important source of water for *Phrynosoma mcallii.* Grant (2005) observed the first known occurrence of rain harvesting in *Phrynosoma mcallii* in two individuals. When rain harvesting, the lizard stands with the venter off the ground and spreads the back. Rain falling on the dorsum moves by capillary action to the corners of the mouth. The lizard makes rhythmic swallowing motions as it ingests the water. Grant (2005) found that the mass of lizards increase after a rainfall event, which is more evidence that this species utilizes rain harvesting as a means of ingesting water (Grant 2005). *Phrynosoma mcallii* is not capable of using its skin to collect water from the environment (Mayhew and Wright 1971).

Habitat

Phrynosoma mcallii occurs in fine packed sand or pavement, overlain with loose, fine sand in areas that are sparse or lacking in vegetation. The species occurs in predominantly sandy flats associated with creosote bush (*Larrea tridentate*), white bursage (*Ambrosia dumosa*), burrobush (*Franseria dumosa*), indigo bush (*Psorothamnus emoryi*), and big galleta (*Hilaria rigida*) (AGFD 2003). The lizards occur at elevations from below sea level to 250 m (AGFD 2005). Vegetation may be an important requirement for oviposition sites (CDFG 1994). *Phrynosoma mcallii* does not normally occur in a number of habitats represented within its geographic range: 1) rocky, mountainous areas, 2) new alluvial areas with sloping terrain, 3) salt flats and mud flats with little or no vegetation, 4) major dune systems, 5) marshes and tamarisk-arrowweed thickets, and 6) agricultural and developed areas (Turner et al. 1980).

There have been several studies conducted correlating *Phrynosoma mcallii* relative abundance to habitat characteristics. Several studies have found high *Phrynosoma mcallii* relative abundance correlated with percent of sandy substrate (Grant 2005, Hodges 1995, Muth and Fisher 1992, Rorabaugh et al. 1987). Wright (2002) found no significant difference between *Phrynosoma mcallii* relative abundance and substrate type (sand, gravel, hardpan). Grant (2005) believes that *Phrynosoma mcallii* may prefer sandy areas, but are not necessarily confined to sandy areas as once believed. Gardner and Foley (2001) observed that *Phrynosoma mcallii* utilized two different types of substrate: compacted sand with a shallow surface of loose grained sands, and loose, small- to medium-grain sand.

Wone and Beauchamp (1995) observed *Phrynosoma mcallii* in hardpan soil covered with gravel and sparse vegetation. Turner and Medical (1982) found high *Phrynosoma mcallii* relative abundance positively correlated with perennial density and diversity. Grant (2005) found no correlation between perennial density and *Phrynosoma mcallii* relative abundance. Hodges (1995) found that plant density, diversity, and percent cover were not correlated with *Phrynosoma mcallii* relative abundance. Rorabaugh et al. (1987) found high *Phrynosoma mcallii* relative abundance in areas where galleta grass dominated. Muth and Fisher (1992) found that *Phrynosoma mcallii* preferred white bursage and indigo bush but avoided creosote bush and coldenia (*Tequilia plicata*).

Beauchamp et al. (1998) found that high *Phrynosoma mcallii* abundance was correlated with large patches of concretions, gravel, silt, and sparse perennial vegetation at the Ocotillo Wells State Vehicular Recreation Area (OWSVRA) in California. High relative abundance was negatively correlated with dense perennial vegetation (Beauchamp et al. 1998). Beauchamp et al. (1998) also found *Phrynosoma mcallii* utilizing mudhills. This suggests that either *Phrynosoma mcallii* has shifted or dispersed to other habitats because of off-highway vehicle use in sandy areas, or the species has a wider habitat preference than previously described (Beauchamp et al. 1998).

The above studies have all found *Phrynosoma mcallii* relative abundance to be correlated with some habitat characteristic; many of these characteristics differ between studies. Some studies have found relative abundance to be correlated with a particular habitat characteristic, while another study has shown that same characteristic to be uncorrelated with relative abundance. While there is a general knowledge of habitat characteristics that may be important to *Phrynosoma mcallii*, there is a lack of data on which characteristic is the most important in determining *Phrynosoma mcallii* relative abundance. More research is needed to determine the most important habitat requirement for this species (Rorabaugh et al. 1987, Turner and Medical 1982). It has always been thought that *Phrynosoma mcallii* were primarily associated with sandy areas, but they have been observed other places. Whether this is a due to disturbance of sandy habitat, or whether *Phrynosoma mcallii* has wider habitat preferences than previously thought is not conclusive. Types of habitat used by *Phrynosoma mcallii* across their range needs to be reevaluated (Beauchamp et al. 1998).

Threats

Historically, *Phrynosoma mcallii* habitat loss occurred due to the creation of the Salton Sea, agricultural conversion, and human expansion. Current threats to *Phrynosoma* *mcallii* include habitat loss from urban and agricultural expansion, pesticide contamination, off-highway vehicle activities, geothermal development, roads, highways, railroads, power lines, military activities, wind turbines, invasive plant species, land disposal, cattle grazing, border patrol activities, sand and gravel extraction and vehicular traffic (AGFD 2003, Bolster and Nicol 1989). Agriculture and urban development have an indirect effect on adjacent *Phrynosoma mcallii* populations up to 450 m away from the project, due to increased predation near development and increased abundance of invasive species (Young and Young 2005). Urban development poses a threat to the species in the Borrego Valley, Coachella Valley, and on the Yuma Mesa near Yuma and San Luis, Arizona (FTHL ICC 2003). Interstate 10, Interstate 8, state routes 86, 78, and 98, Coachella Canal, and Borrego Valley bisect *Phrynosoma mcallii* habitat and act as barriers to movement (FTHL ICC 2003).

Hodges (1997) estimated that 63,129 acres (31.10%) of historical *Phrynosoma mcallii* habitat in Arizona has been lost due to agricultural conversion (35,520 acres), urban development (22,624 acres), and military use (5,082 acres). Hodges (1997) estimated that 1,112,640 acres (50.20%) of historical *Phrynosoma mcallii* habitat in California has been lost due to flooding of the Salton Sea, agricultural conversion, urban development, military activities, and fire. Hodges estimated that 20,393 acres of habitat is currently threatened by urban development in Riverside County, the Yuma Area Service Highway, Arizona State Prison, a large regional landfill, small local landfills, and military activities.

Threats to *Phrynosoma mcallii* in the LCR MSCP planning area occur in the 5-mile zone, located in the Yuma Desert southeast of Yuma, Arizona, and west of the Barry M. Goldwater Range (USBR 1997). Activities in this area include the operation and maintenance of the 242 well field, operation of the YDP sludge disposal site, and maintenance of the canal used for delivery of water to Mexico (Bureau of Reclamation 1996). There are approximately 40 records for *Phrynosoma mcallii* within this zone (USBR 1997). Reclamation has preserved 16,000 acres in the Yuma Desert Management Area (USBR 1997). Future threats to *Phrynosoma mcallii* in the LCR MSCP planning area include operation of vehicles and equipment necessary to maintain and replace facilities and infrastructure or roads and other infrastructure required to install or maintain restored habitat (MSCP HCP 2004).

Vehicular traffic is a direct threat to *Phrynosoma mcallii* populations; many individuals are killed by vehicles when on the road. The primary defense behavior of *Phrynosoma mcallii* is to remain motionless and rely on camouflage to avoid predation, which makes them more susceptible to fatalities by vehicles because they remain on the road as the vehicle approaches (Young and Young 2000.) A paved road, with even moderate amounts of traffic, would negatively impact any population of *Phrynosoma mcallii* within at least 500 m of either side of the road, with severe impacts within 250 m of the road (Young and Young 2000). Every kilometer of road would potentially impact 100 hectares of habitat.

Gardner et al. (2004) designed fencing that was successful in keeping *Phrynosoma mcallii* off roads. The fence was constructed with four rolls (30.5 m long, 90 cm high) of hardware cloth with 6.5-mm (1/4-inch) mesh, buried to a depth of 15 cm and attached with cable ties to rebar supports at 2.5 m intervals. Fences were located five meters from the edge of roads (Gardner et al. 2004). Suggested improvements to the design include using wire instead of hardware cloth and using posts to support seams between wire rolls (Gardner et al. 2004). Research has been conducted to design crossing structures under roads using different size culverts and sky lights (Painter and Ingraldi 2005). Other strategies to reduce fatalities by vehicles are to allow vehicle traffic on roads only during the *Phrynosoma mcallii* hibernation period (November through March), restrict traffic to the heat of the day or after dark (1200 to 1600 h and 2000 to 0500 h), or prohibit traffic when the temperatures are between 25°C and 35°C (Young and Young 2000).

Off-highway vehicle (OHV) usage is an increasingly popular activity that takes place in *Phrynosoma mcallii* habitat. OHV usage may pose direct threats (mortality by being run over) or indirect threats (destroying ant mounds, affecting vegetation, compacting soil) to *Phrynosoma mcallii* populations. Studies on impacts of OHV use on *Phrynosoma mcallii* are incomplete and inconclusive (FTHL ICC 2003). Wright (1993) found that *Phrynosoma mcallii* relative abundance varied significantly between use classifications (open, limited) but not between levels of use in the Algodones Dunes. Wright (2002) and Wright and Grant (2003) found no consistent relationship between vehicle impacts and *Phrynosoma mcallii* detection rates in the Yuha Basin. Grant (2005) found that OHV activity did not directly effect *Phrynosoma mcallii* populations during hibernation, which is the main OHV season. Setser (2004) did not observe any direct *Phrynosoma mcallii* mortality due to OHV activity. Rates and direction of *Phrynosoma mcallii* movement differed significantly before OHV races versus after OHV races but the impact that those findings have to the species is unknown (Nicola and Lovich 2000). *Phrynosoma mcallii* may have shifted habitat use from sandy areas to other areas (concrete, mudhills, gravel, silt) at OWSVRA due to OHV use (Beauchamp et al. 1998). More research dealing with direct and indirect effects of OHV activity on *Phrynosoma mcallii* is needed.

The United States Border Patrol conducts patrols and rescues near the international border that sometimes involve cross-country travel. Border patrol activities in *Phrynosoma mcallii* habitat have greatly increased since 1997 (Rorabaugh *in* FTHL ICC 2003). Border Patrol activities may have contributed to the dramatic increase of OHV tracks in the Yuma Desert, Yuha Desert, and West Mesa Management Areas (Wright 1993, Wright 2002, Rorabaugh et al. 2002 *in* FTHL ICC 2003).

The population viability analysis conducted by the flat-tailed horned lizard conservation team suggested that modest increases in mortality (even age-specific) negatively affect population over a 100-year time period (Foreman 1997). Activities that cause direct mortality, such as vehicular traffic or OHV activity, should be limited.

The primary predators of *Phrynosoma mcallii* are the round-tailed ground squirrel (*Spermophilus tereticaudus*) and loggerhead shrike (*Lanius ludovicianus*). Other predators of *Phrynosoma mcallii* include the grasshopper mice (*Onychomys* spp.), American kestrel (*Falco sparverius*), common raven (*Corvus corax*), burrowing owl

(*Athene cunicularia*), snakes, and feral cats and canids (AFGD 2003, Duncan et al. 1994; Natureserve 2005).

Flat-tailed Horned Lizard Rangewide Management Strategy

A rangewide management strategy for *Phrynosoma mcallii* was completed in 1997 and revised in 2003 (Foreman 1997, FTHL ICC 2003). The strategy has nine categories of planning actions that conservation measures fall under: 1) delineate and designate five flat-tailed horned lizard Management Areas and one Research Area, 2) define and implement management actions necessary to minimize loss or degradation of habitat, 3) rehabilitate damaged and degraded habitat within Management Areas, 4) attempt to acquire all private lands within Management Areas, 5) maintain or establish effective habitat corridors between naturally adjacent populations, 6) coordinate activities and funding among the participating agencies and Mexican agencies, 7) promote the purposes of the strategy through law enforcement and public education, 8) encourage and support research to promote conservation of flat-tailed horned lizard and desert ecosystems, and 9) monitor habitat quality and population trends in the management areas (Foreman 1997). Annual accomplishments and proposed actions are described in FTHL ICC (2003 and 2006).

Survey Methods

A variety of methods have been used to estimate *Phrynosoma mcallii* relative abundance throughout its range. Beauchamp et al. (1998) surveys consisted of belt transects spaced 20 m apart that were surveyed four times between June and July. Observers counted all *Phrynosoma mcallii* and *Phrynosoma mcallii* scat greater than 5.5 mm in diameter (Beauchamp et al. 1998). Hodges (1995) surveys consisted of belt transects spaced 5 m apart that were surveyed three times between April and August. Observers counted all *Phrynosoma mcallii* and *Phrynosoma mcallii* scat greater than 5.5 mm in diameter (Hodges 1995). Turner and Medica (1982) and Rorabaugh et al. (1987) used the section search procedure to determine *Phrynosoma mcallii* relative abundance. A section search is a 1-hour walk through a 2.50-km^2 area, where observers count *Phrynosoma mcallii* and *Phrynosoma mcallii* scat (Turner and Medica 1982). Turner and Medical (1982) suggested that relative abundance estimates should be based on several section searches per township. The Bureau of Land Management (BLM) used 2.5-mile triangular transects, where observers walked transects in 1 hour and counted all lizards and scat greater than 5.5 mm (Olech undated, Wright 1993, Wright 2002). Wright (2002) suggested that at least 55 triangular transects should be conducted per area assessed for *Phrynosoma mcallii* abundance. Hodges (1995) found that when BLM triangular survey results were compared to more intensive survey method results utilizing belt transects, there was a significant difference in density classifications (poor, low, medium, high) between survey methods.

All of these methods relied upon systematic counts of scat as an accurate assessment of *Phrynosoma mcallii* abundance. *Phrynosoma mcallii* produce large, visible scats that consist mostly of ant parts (Rorabaugh 1987). Scat counts are an attractive alternative to direct enumeration of *Phrynosoma mcallii* because this species is difficult to locate and scat counts are simple, cost effective, and yield quantitative results (Rorabaugh 1994).

There have been questions of whether scat count indexes are a reliable indicator of *Phrynosoma mcallii* abundance and distribution. Annual differences in scat counts may be influenced by the rate of scat production per lizard rather than the abundance of *Phrynosoma mcallii* (Rorabaugh 1994). Scat counts are affected by wind, heavy rains, and observer bias (Rorabaugh 1994). Scats may have a clumped distribution within *Phrynosoma mcallii's* home range; they may be more of an indicator of overnight location rather than habitat utilization (Muth and Fisher 1992). Young and Young (2000) suggest that the effects of climate on scat production and scat size may cause too much variance to ever be able to detect true population trends using scat surveys.

Several studies have concluded that *Phrynosoma mcallii* scat counts and number of *Phrynosoma mcallii* were not correlated (Beauchamp et al. 1998, Hodges 1995). Wright (1993) found a significant correlation between *Phrynosoma mcallii* and *Phrynosoma mcallii* scat per hour but regression analysis showed that scat per hour is a poor predictor of the actual number of *Phrynosoma mcallii* sightings. *Phrynosoma mcallii* scats are only distinguishable from *Phrynosoma platyrhinos* scats if they are greater than 5.5 mm in diameter (Muth and Fisher 1992). Rorabaugh (1994) suggested that scat counts should be used cautiously and combined with habitat evaluations and locality of records of *Phrynosoma mcallii* in assessing the importance of habitat areas for this species. Muth and Fisher (1992) recommend that scat counts should be used only to determine relative abundance and not to infer habitat quality. Survey methods using scat count indices are crude, do not give the actual population size, and have low sensitivity to changes in population size (Wright and Grant 2003).

Population trends have been difficult to detect across *Phrynosoma mcallii's* range due to inconsistent monitoring protocols and the inaccuracy of scat counts (Foreman 1997). Mark and recapture methods combined with monitoring changes in distribution with presence/absence survey should increase sensitivity in detecting future trends (FTHL ICC 2003). FTHL ICC (2003) created a standardized mark and recapture monitoring protocol based on Wright and Grant (2003), and a standardized distribution monitoring protocol.

Mark and recapture methods have been used to assess *Phrynosoma mcallii* density (Turner and Medica 1982, Wone et al. 1994, Wright and Grant 2003, Grant 2005). Wone et al. (1994) found that a 400 by 400 m plot sampled by twenty 400-m long belt transects spaced 20 m apart produced statistically valid samples to be used in a mark and recapture study. Wone et al. (1994) made the following suggestions with regards to mark and recapture studies: 1) mark *Phrynosoma mcallii* by filing a notch on their occipital horn, 2) juvenile *Phrynosoma mcallii* should not be marked due to ontogenetic changes in their occipital horns, and 3) Sequential Bayes Algorithm should be used to analyze mark-recapture data to estimate population size. Grant (2005) used closed mark recapture and distance sampling methods to estimate population size of *Phrynosoma mcallii*. Closed mark-recapture methods were compromised from a lack of geographic closure; suggested improvements include larger plots, enclosed plots, or application of Pollock's robust design (Grant 2005).

Other suggestions for improving the closed mark and recapture method were the use of a covariate to take observer bias into account and to increase the sampling fraction of all areas (Grant 2005). Grant (2005) suggested that further distance sampling studies should try to estimate detection probability on the line. Grant (2005) stressed the importance of sample size in mark and recapture methods and distance sampling methods. Occupancy estimation is another technique recently used to monitor *Phrynosoma mcallii* populations; it is less time consuming than mark and recapture, allowing for more area to be covered (BLM El Centro 2005). Another technique discussed in the literature is to combine habitat protection with presence/absence surveys for *Phrynosoma mcallii* (Young and Young 2000). Young and Young (2000) felt that this technique is more feasible and more likely to ensure persistence over time than monitoring changes in density. Presence/absence surveys could delineate the habitat where these lizards occur and would indicate where large, continuous chunks of critical habitat are. Loss of habitat and/or changes from presence to absence over a wide area would signal problems.

LCR MSCP Conservation Measures

FTHL1—Aquire and protect 230 acres of existing unprotected occupied flat-tailed horned lizard habitat.
Consistent with the mitigation measures identified in the Flat-Tailed Horned Lizard Rangewide Management Strategy (FTHL ICC 2003), the LCR MSCP will acquire and protect 230 acres of unprotected occupied flat-tailed horned lizard habitat. The acquired habitat will be transferred to an appropriate management agency for permanent protection of habitat for the species.

FTHL2—Implement conservation measures to avoid or minimize take of flat-tailed horned lizard.
Reclamation will continue to implement measures to avoid or minimize take of flat-tailed horned lizard. These measures would include worker education programs and other procedures as described in the 1997 BO (U.S. Fish and Wildlife Service 1997) and are in accordance with the 2003 Flat-tailed Horned Lizard Interagency Coordinating Committee recommendations for the species**.**

LCR MSCP Research and Monitoring Needs

LCR MSCP conservation measures for flat-tailed horned lizard consist of acquiring unprotected occupied habitat and implementing conservation measures to avoid or minimize take. The California OHV Division headquarters and the BLM-El Centro office maintain a list that prioritizes parcels for acquisition. Priority is based on habitat quality, purchasing difficulty, and level of protection from threats to *Phrynosoma mcallii* (FTHL ICC 2003, 2006). Land in Arizona adjacent to the Yuma Desert Management area can be acquired. Land that acts as corridors between the West Mesa and Yuha Desert Management Areas and between the West Mesa Management Area and Ocotillo Wells Research Area can be acquired (FTHL ICC 2003, 2006). Habitat quality and number of threats to *Phrynosoma mcallii* should be assessed on land to be acquired. OHV use and vehicular traffic are major threats to *Phrynosoma mcallii* and should be minimal on land acquired. Other previous mention threats to *Phrynosoma mcallii* should also be minimal on land acquired.

Acquired habitat should be protected in a manner consistent with mitigation measures detailed in the Flat-tailed Horned Lizard Rangewide Management Strategy (FTHL ICC 2003). These conservation measures include: 1) limiting land use authorizations that would cause surface disturbances, 2) establishing flat-tailed horned lizard fencing, according to the protocol outlined in Appendix 7 of the Rangewide Management Strategy, as a barrier along roads and around project areas, 3) conduct removal surveys according to the protocol outlined in Appendix 7 of the Rangewide Management Strategy in project areas, 4) restore the area once project is complete, and 5) limit vehicle access, OHV activity, military activities and camping to designated areas (FTHL 2003).

Other Research and Monitoring Opportunities

Habitat acquisition will be conducted using priorities established by the Flat-tailed Horned Lizard Interagency Coordinating Committee. Research and monitoring will not be conducted on the flat-tailed horned lizard during LCR MSCP implementation. Additional research could be conducted by other conservation programs, including:

- A general understanding of *Phrynosoma mcallii* habitat requirements are known, but limiting factors affecting *Phrynosoma mcallii* density need to be determined, including the presence of harvester ant colonies, substrate type, perennial plant diversity, perennial plant composition, and perennial plant density.

- *Phrynosoma mcallii* has been observed in different habitat types in disturbed areas (Beauchamp et al. 1998). Does *Phrynosoma mcallii* habitat use shift in disturbed environments or does *Phrynosoma mcallii* have a broad habitat preference (Beauchamp et al. 2003)?

- The impacts of OHV use on *Phrynosoma mcallii* are incomplete and inconclusive (FTHL ICC 2003). More research needs to be done on the direct and indirect effects of OHV use on *Phrynosoma mcallii* populations. Research topics may include the effect of OHV use on the movement and home ranges of *Phrynosoma mcallii* (Nicola and Lovich 2000), the indirect effects that OHV use has on *Phrynosoma mcallii* (i.e., vegetation destruction, depletion of food sources), the survival of vegetation crushed by OHV or regular vehicles that travel off road such as border patrol vehicles (Piest 2002), and the changes in *Phrynosoma mcallii* home range response to a depleted food source (Young 1999).

- Studies need to be conducted on the genetic variation across *Phrynosoma mcallii* range. The five core populations of *Phrynosoma mcallii* are relatively isolated, which has caused certain populations to have diverged more than others (Young and Young 2000). Hybrids between *Phrynosoma mcallii* and *Phrynosoma platyrhinos* have been reported in Ocotillo, California, and on the Barry M. Goldwater Range in Yuma, Arizona (Foreman 1997, AFGD 2003). Genetic analysis also needs to be done to determine whether there is a zone of hybridization between *Phrynosoma mcallii* and *Phrynosoma platyrhinos.*

- Studies need to be conducted on the effects of certain threats on *Phrynosoma mcallii* such as paved roads and highways, OHV use and associated activities, geothermal development, pesticide use, predation, nonnative plants, fire, and wind turbines.

Literature Cited

Arizona Game and Fish Department. 2003. Unpublished abstract compiled and edited by the Heritage Data Management System, Arizona Game and Fish Department, Phoenix, Arizona. 7 p.

Beauchamp, B., B. Wone, S. Wone, and M. Kutilek. 1998. Habitat use of the flat-tailed horned lizard (*Phrynosoma mcallii*) in a disturbed environment. Journal of Herpetology 32(2):210-216.

Bolster, and K. Nicol. 1989. The status of the flat-tailed horned lizard (*Phrynosoma mcallii)* in California. Report to California Department of Fish and Game, Sacramento, California.

Bureau of Land Management. 2005. Flat-tailed Horned Lizard Monitoring Report 2005. El Centro Field Office, California.

Bureau of Reclamation. 1996. Description and Assessment of Operations, Maintenance, and Sensitive Species of the Lower Colorado River. U.S Bureau of Reclamation, Lower Colorado Region. Final Biological Assessment Prepared for U.S. Fish and Wildlife Service and Lower Colorado River Multi-Species Conservation Program.

California Department of Fish and Game. 1994. Amphibian and Reptile Species of Special Concern in California, Flat-tailed Horned Lizard. Habitat Conservation Planning Branch, California Department of Fish and Game. Accessed on 9/08/2005. http://www.dfg.ca.gov/hcpb/cgi-bin/read_one.asp.

Carlson, B.A., and W.W Mayhew. 1988. A petition to the state of California Fish and Game Commission (*Phrynosoma mcallii*). University of California Riverside.

Duncan, R.B., T.C. Esque, and K.L. Echold. 1994. *Phrynosoma mcallii* (flat-tailed horned lizard) predation. Herpetological Review 25:68.

Fisher, M., W. Hodges, A. Holycress, J. Howland, J. Lovich, P. Medica, A. Muth, J. Rorabaugh, F. Turner, B. Wone, and L. Vitt. 1998. Flat-tailed horned lizard, *Phrynosoma mcallii*, population viability analysis: Implications for conservation strategies and research priorities. Unpublished. 20 p. plus appendices.

Flat-tailed Horned Lizard Interagency Coordinating Committee. 2003. Flat-tailed horned lizard rangewide management strategy, 2003 revision. 78 p. plus appendices**.**

Flat-tailed Horned Lizard Interagency Coordinating Committee. 2006. Annual Progress Report: Implementation of the Flat-tailed Horned Lizard Rangewide Management Strategy. March 2006.

Foreman, L.D. (editor). 1997. Flat-tailed horned lizard rangewide management strategy. Report of interagency working group. 61 p. plus appendices.

Funk, R.S. 1981. *Phrynosoma mcallii* (Hallowell) flat-tailed horned lizard. Catalogue of American Amphibians and Reptiles:281.1-281.2

Gardner, T.J., D.H Foley III, and E.D Brodie Jr. 2004. Barrier fences prevent road mortalities in the flat-tailed horned lizard (*Phrynosoma mcallii*). Herpetological Review 35(3):250-251.

Grant, T.J. 2005. Flat-tailed Horned Lizards (*Phrynosoma mcallii*): Population of Size Estimation, Effects of Off-highway Vehicles, and Natural History. M.S. Thesis. Colorado State University, Fort Collins.

Hodges, W.L. 1995. *Phrynosoma mcallii* occurrence in Arizona. Department of Zoology, University of Texas at Austin. Arizona Game & Fish Heritage Fund Contract Q95-15-K.

Hodges, W.L. 1997. Assessing *Phrynosoma mcallii* (flat-tailed horned lizard) habitat loss in Arizona and California. Unpublished contract for the University of Texas, Austin, Texas.

Howland, J., and J. Rorabaugh. 1996. A saving strategy for the flat-tailed horned lizard. Bajada, Vol. 4(2). National Biological Service, Cooperative Park Studies Unit, University of Arizona, Tucson.

Howard, C.W. 1974. Comparative reproductive ecology of horned lizards (Genus *Phrynosoma*) in the southwestern United States and northern Mexico. Journal of the Arizona Academy of Science 9:108-116.

Lower Colorado River Multi-Species Conservation Program. 2004. *Lower Colorado River Multi-Species Conservation Program, Volume II: Habitat Conservation Plan* Final. December 17. (J&S 00450.00.) Sacramento, California.

Mayhew W., and S.J. Wright. 1971. Water impermeable skin of the lizard *Phrynosoma mcallii.* Herpetologica Volume 27:8-11.

Mayhew W. 1965. Hibernation in the horned lizard, *Phrynosoma mcallii*. Comparative biochemistry and physiology 16:103-119.

Miller, P.A 1999. Home range of the flat-tailed horned lizard *Phrynosoma mcallii.* Utah State University, Logan.

Muth A., and M. Fisher. 1992. Development of baseline data and procedures for monitoring populations of the flat-tailed horned lizard, *Phrynosoma mcallii.* California Department of Fish and Game.

NatureServe. 2005. NatureServe Explorer: An online encyclopedia of life (web application). Version 4.5. NatureServe, Arlington, Virginia. Accessed on September 8, 2005. http://www.natureserve.org/explorer.

Nicola, N.C., and J.E Lovich. 2000. Preliminary observations of the behavior of male, flat-tailed horned lizards before and after an off-highway vehicle race in California. California Fish and Game 86:208-212

Norris, K.S. 1949. Observations on the habits of the horned lizard *Phrynosoma mcallii.* Copeia 1949:176-180.

Olech, L.A. 1986. Status of the flat-tailed horned lizard (*Phrynosoma mcallii)* on Bureau of Land Management administered land in California. Bureau of Land Management, California Desert District, El Centro Resource Area.

Painter, M.L., and M. Ingraldi. 2005. Use of simulated highway underpass crossing structures by flat-tailed horned lizards (*Phrynosoma mcallii*). Progress Report. Research Branch, Arizona Game and Fish Department, Flagstaff, Arizona.

Piest L., and B. Knowles. 2002. An analysis of the status and land ownership of flat-tailed horned lizard habitat in Arizona. Unpublished Arizona Game and Fish Department Report. Yuma, Arizona.

Rado, T.A. 1981. Analysis of actual and potential loss of flat-tailed horned lizard (*Phrynosoma mcallii*) habitat. Bureau of Land Management, Sacramento, California.

Repp, R.A. 1999. The flat-tailed horned lizard, *Phrynosoma mcallii:* A lizard imperiled. Vivarium 10:42-43.

Rodrigues, R. 2002. Evaluation of the status, distribution and development of education/interpretation materials of the flat-tailed horned lizard, *Phrynosoma mcallii* in Mexico. Unpublished report. Centro Intercultural de Estudioes de Desiertos y Oceanos.

Rorabaugh, J.C., C.L. Palermo, and S.C. Dunn. 1987. Distribution and relative abundance of the flat-tailed horned lizard (*Phrynosoma mcallii*) in Arizona. The southwestern naturalist 32:103-109.

Rorabaugh, J. 1994. An analysis of scat counts as a survey method for the flat-tailed horned lizard (*Phrynosoma mcallii).* Unpublished report. U.S. Fish and Wildlife Service, Phoenix, Arizona.

Rorabaugh, J. 1996a. Surveys for flat-tailed horned lizard at Pinta Sands, Cabeza Prieta National Wildlife Refuge, Arizona, 3-5 May 1996. Memorandum. U.S. Fish and Wildlife Service, Phoenix, Arizona.

Rorabaugh, J. 1996b,c. Surveys for the flat-tailed horned lizard, *Phrynosoma mcallii* at the Salton Sea Test Base, Imperial County, California. Report to the Department of the Navy, Southwest Division, San Diego, California.

Rorabaugh, J. 1996d. Surveys for the flat-tailed horned lizard, *Phrynosoma mcallii*, at the Carrizo Impact Area, Imperial County, California. Report to the Department of the Navy, Southwest Division, San Diego, California.

Rorabaugh, J. 1997. Surveys for flat-tailed horned lizard at Pinta Sands, Cabeza Prieta National Wildlife Refuge, Arizona, 5-7 May 1997. Memorandum. U.S. Fish and Wildlife Service, Phoenix, Arizona.

Rorabaugh, J., M. Coffeen, and L. Piest. 2002. Human disturbance in the flat-tailed horned lizard Yuma Desert Management Area. Unpublished Report. 14 p.

Sester, K. 2004. Natural history, demography, and home range characteristics of a southern California population of *Phrynosoma mcallii* inhabiting atypical habitat. Unpublished Masters Thesis, Utah State University, Logan.

Turner, F.B., P.A. Medica, and H.O. Hill. 1978. The status of the flat-tailed horned lizard *Phrynosoma mcallii* at 9 sites in Imperial and Riverside counties, California. Report to Bureau of Land Management, El Centro, California.

Turner, F.B., J.C. Rorabaugh, E.C. Nelson, and M.C Jorgenson. 1980. A survey of the occurrence and abundance of the flat-tailed horned lizard (*Phrynosoma mcallii*) in California. Laboratory of Nuclear Medicine and Radiation Biology, University of California, Los Angeles.

Turner, F.B., and P.A. Medica. 1982. The distribution and abundance of the flat-tailed horned lizard (*Phrynosoma mcallii*). Copeia 1982:815-823.

U.S. Fish and Wildlife Service. 1997. Final Biological and Conference Opinion on Lower Colorado River Operations and Maintenance—Lake Mead to Southerly International Boundary. U.S. Fish and Wildlife Service, Albuquerque, New Mexico.

U.S. Fish and Wildlife Service. 2002. News Release, January 3, 2002. Accessed on 9/08/2005 http://www.fws.gov/arizonaes.

U.S. Fish and Wildlife Service. 2003. Summary of Management Actions for the Flat-tailed Horned Lizard (*Phrynosoma mcallii*). Accessed on 9/08/2005 http://www.fws.gov/arizonaes.

U.S. Fish and Wildlife Service. 2006. News Release, April 21, 2006. Accessed on 5/09/2006 http://www.fws.gov/arizonaes.

U.S. Fish and Wildlife Service. 2006. Federal Register Vol. 70, No. 234, December 7, 2005. Accessed on 5/09/2006 http://www.fws.gov/arizonaes.

U.S. Fish and Wildlife Service. 2006. Federal Register Vol. 71, No. 124, June 28, 2006. Accessed on 8/28/2006 http://www.fws.gov/arizonaes.

Wone B., B. Beauchamp, E.G. Olson, and B.O. Wolf. 1991. Occurrence and distribution of the flat-tailed horned lizard, *Phrynosoma mcallii*, at Ocotillo Wells State Vehicular Recreation Area and the acquisition area, California, based upon a survey of scats. Contract Report No. SG-0514, California Department of Fish and Game.

Wone, B., B. Beauchamp, and M. Kutilek. 1994. Development of methods for monitoring population trends of *Phrynosoma mcallii* at Ocotillo Wells State Vehicular Recreation Area. California Report to California Department of Parks and Recreation, Contract No. C9314012. 21 p.

Wone, B., and B. Beauchamp. 1995. Observations of the escape behavior of the horned lizard *Phrynosoma mcallii*. Herpetological Review 26:132

Wone, B., and B. Beauchamp. 2003. Movement, home range and activity patterns of the horned lizard, *Phrynosoma mcallii*. Journal of Herpetology 37:679-686.

Wright, G.R. 1993. Flat-tailed horned lizard status report—September 1993. BLM Report, El Centro Resource Area, California. 59 p. plus appendices and maps

Wright, G.R. 2002. Flat-tailed horned lizard monitoring report. Bureau of Land Management Report. El Centro Resource Area, California. 55 p.

Wright, G., and T. Grant. 2003. Flat-tailed horned lizard monitoring report. Bureau of Land Management Report. El Centro Resource Area, California. 55 p.

Young, K.V. 1999. Scientific study of the flat-tailed horned lizard, *Phrynosoma mcallii*, at Ocotillo Wells SVRA, 1998 Field Season. Unpublished Report. California State Parks Off-highway Motor Vehicle Division, Sacramento, California.

Young, K.V., and A.T. Young. 2005. Indirect effects of development on the flat-tailed horned lizard. Final Report submitted to Arizona Game and Fish Department, Yuma. 11 p.

RELICT LEOPARD FROG
(*Rana onca*)

Legal Status

Platz (1984) suggested that *Rana onca* should be considered extinct. The last known specimen of *Rana onca* was seen in Utah in the 1950s and is believed to be extirpated in the state. *Rana onca* was rediscovered in 1991 in parts of its historical range (Black Canyon/Virgin River) through southern Nevada and northwestern Arizona (Bradford et al. 2004). Seven populations of *Rana onca* were found in three distinct areas (Bradford et al. 2004). *Rana onca* was listed as a candidate species on 4 May 2004 (USFWS 2004). A petition to list *Rana onca* as an endangered species was received by the U. S. Fish and Wildlife Service (USFWS) on 9 May 2002 (USFWS 2004). Natureserve, a nonprofit organization that ranks species on their relative imperilment, ranked *Rana onca* as critically imperiled across its range. Natureserve classifies critically imperiled species as species that have a very high risk of extinction due to extreme rarity, steep declines, or other factors (Natureserve 2005). *Rana onca* is listed as a species of special concern in Arizona.

Distribution

Historical Range

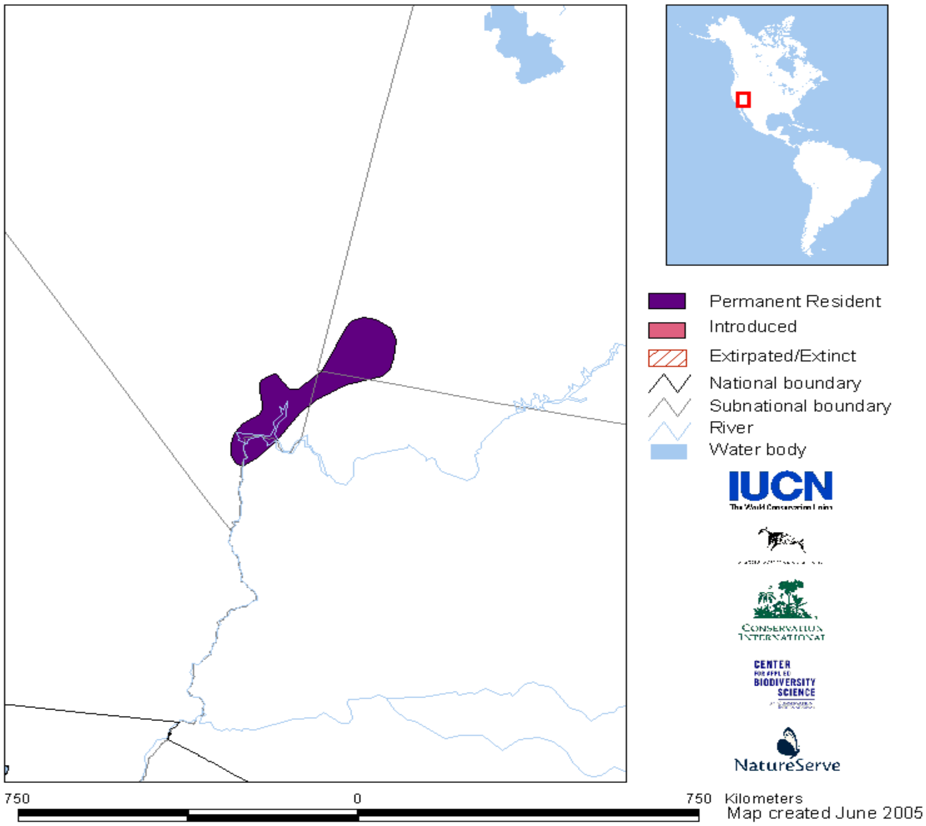
Rana onca historically occurred within the Virgin River drainage downstream from the vicinity of Hurricane, Utah, along the Muddy River drainage in Nevada, and along the Colorado River from its confluence with the Virgin River downstream to the Black Canyon area below Lake Mead, Nevada and Arizona (RLFTC 2005, Jaeger et al. 2001, Bradford et al. 2004). *Rana onca* was historically found in Nevada around the Overton Arm of what is now Lake Mead, along the Muddy River and Meadow Valley Wash northwest of the Overton Arm, and within the Black Canyon along the Colorado River. *Rana onca* was historically found in Utah from the vicinity of Hurricane, Washington County, downstream through the Virgin River Valley, at elevations between 370 and 760 m (Center for Biological Diversity 2002). Historical abundance is unknown (Bradford and Jennings 2005).

The Arizona Game and Fish Department (AGFD) conducted surveys within the historical range of *Rana onca* in Arizona. Limited surveys were conducted before 1997 in which *Rana onca* was detected at one site near Littlefield, Arizona. Surveys were conducted from February to November in the years 2001, 2002, and 2003 at 74 locations; no *Rana onca* were found. Six of the 74 sites fit AGFD’s description of suitable habitat, although non-native predators would have to be removed from three of the sites (Blomquist et al. 2003). Platz (1984) conducted surveys in historical habitat around Las Vegas and along the Virgin River northeast of Las Vegas to the vicinity of St. George, Utah. No *Rana onca* were found. *Rana onca* was rediscovered at Corral and Bluepoint springs in 1991. Bradford et al. (2004) conducted targeted surveys for *Rana onca* at 66 locations across its historical range between 1991 and 2001. Bradford et al. (2004) conducted visual estimate and mark and recapture surveys on extant populations of *Rana onca* between 1991 and 2001 to determine population size and structure. Bradford et al. (2004) conducted visual encounter surveys at Blue Point, Rogers, and Corral springs from July 1993 to November 1996, in the Black Canyon area (Boy Scout, Saltcedar and Bighorn Sheep springs) from 1997 to 2001, and at Reber Spring from 1998 to 2001. Bradford et al. (2004) conducted mark-recapture studies at Blue Point Spring in 1995 and 1996 and at Bighorn Sheep Spring in 2001. Members of the relict leopard frog conservation team conduct annual surveys for *Rana onca* of occupied and historical unoccupied habitat (RLFTC 2005). They also conduct surveys of potential translocation sites in *Rana onca* historical habitat (RLFTC 2005).

Current Range

Extant populations of *Rana onca* were detected at seven sites in three distinct areas during the 1990s: 1) Overton Arm of Lake Mead, Clark County, Nevada (Blue Point, Rogers, and Corral springs—3.6 km in length), 2) Black Canyon near the Colorado River below Lake Mead, Clark County, Nevada (Boy Scout, Salt Cedar, and Bighorn Sheep springs—5.1 km in length), and 3) adjacent to the Virgin River four kilometers northeast of Littlefield, Mohave County, Arizona (Reber Spring) (Bradford et al. 2004, Center for Biological Diversity 2002). The populations at Corral and Reber springs were extirpated in 1995 and 1998, respectively. Probable causes for these extirpations were emergent vegetation encroachment and the presence of American bullfrogs (*Rana catesbeiana*) (Bradford et al. 2004). Bradford et al. (2004) estimated that the total number of frogs at all sites, based on mark-recapture data, VESs, and extent of habitat were 1100 frogs: 330 in the Overton Arm area and 747 in the Black Canyon area (Bradford et al. 2004). The largest population was present at Bighorn Sheep Spring, which contained 637 frogs (Bradford et al. 2004). Thirty-seven frogs were present at Blue Point Spring. The mark-recapture study at Blue Point Spring in 1996-1997 estimated that *Rana onca* had an annual survival rate of 27% for those years (Bradford et al. 2004). The U.S. Fish and Wildlife Service (USFWS) estimated that the current distribution is less than 20% of the historical distribution (USFWS 2004).

Figure 1: Range of *Rana onca*



Life History

General Description

Henry Crecy Yarrow collected the first specimen of *Rana onca* in 1872 in the Virgin River near the vicinity of St. George, Washington County, Utah. The specimen was a single adult female described by Edward Drinker Cope. *Rana onca* is a true frog (family Ranidae) in the *Rana pipiens* complex (leopard frogs) (Jennings 1988). The following traits distinguish *Rana onca* from other species in the *Rana pipiens* complex: 1) short, indistinct, dorsolateral folds that extend one half to three quarters down the dorsum, 2) generally shortened legs, 3) incomplete supralabial stripe, 4) upper surfaces of the thighs spotted, and 5) venter in the region of the groin is yellow to orange. Males have an enlarged tympana, paired vocal sacs, and no vestigal oviducts (Jennings 1988, AGFD 2003, Amphibiaweb 2005, Natureserve 2005). Males are less spotted, more uniform in color, and are smaller in size than the females (Jennings 1988). Fully developed tadpoles reach 85 mm in length and have a greenish olive dorsum, a heavily mottled, pale green-yellow tail, and a light venter (Amphibiaweb 2005). Wright and Wright (1949) described the call of *Rana onca* to be shorter and not as loud as *Rana pipiens* (Platz 1984).

Breeding

Rana onca breeds in January through April, with peak oviposition occurring in February and March. Signs of oviposition have also been reported in November and calling has been heard in June (AFGD 2004, Center for Biological Diversity 2002, Natureserve 2005, Bradford and Jennings 2005). Oviposition is known to vary between sites (Bradford and Jennings 2005). *Rana onca* deposits egg masses in clusters of up to 250 eggs (AFGD 2003, Center for Biological Diversity 2002, NatureServe 2005, Bradford and Jennings 2005). Males reach reproductive maturity at 42 mm in length measured from snout to vent (SVL) (Bradford and Jennings 2005). Malfatti (1988) was able to create conditions that allowed *Rana onca* to breed in captivity. The life history of *Rana onca* especially in regards to breeding and feeding habits has not been extensively studied and much is unknown (Bradford and Jennings 2005).

Diet

Adults are invertivourus, feeding on insects, spiders, crustaceans, and vertebrates. Larvae are herbivorous feeding on algae, organic debris, and plant tissue (AGFD 2003, Center for Biological Diversity 2002, NatureServe 2005).

Habitat

Rana onca inhabits permanent streams, springs, and spring-fed wetlands below 720 m in elevation that have constant water temperatures between 16 and 55°C (Bradford et al. 2004, Center for Biological Diversity 2002). Historic *Rana onca* habitat included sites with some submerged, emergent, or perimeter vegetation that supports an adequate amount of food resources (Center for Biological Diversity 2002, Jennings and Hayes 1994). Lentic systems at least 400 m^2 in area and lotic systems at least 400 m in length with a depth of 1 m are defined as suitable habitat by the Arizona Game and Fish Department (AGFD). Habitat must have adequate heterogeneity to provide cover and oviposition sites and be free of nonnative predators (Blomquist et al. 2003). Platz (1984) described *Rana onca* historical habitat as sites with permanent cold water and stream pools that are 12 to 16 inches deep.

The five sites currently inhabited by *Rana onca* are characterized by spring systems with largely unaltered hydrology and no introduced *Rana catesbeiana* or game fishes (Bradford et al. 2004). Excessive emergent vegetation of native and non-native species is believed to be a threat to *Rana onca* (RLFTC 2005). Bradford and Jennings (2005) observed that adults prefer relatively open shorelines where dense vegetation does not dominate. Vegetation encroachment has believed to be the major cause of *Rana onca* being extirpated from Corral and Reber springs (Bradford and Jennings 2005). This hypothesis has not been tested and more information about *Rana onca's* habitat requirements in regards to vegetation encroachment is needed. A microhabitat study using radio telemetry was conducted on the *Rana onca* refugium at the Boulder City Wetlands for the purpose of determining microhabitat preference of adult frogs, but could not be completed due to the unsuccessful translocation of *Rana onca* (Ulepic 2002). The University of Las Vegas is currently conducting two studies on habitat requirements of *Rana onca* at Blue Point Spring. The first study began in 2003, and tested the hypothesis that the preferred habitat for adult *Rana onca* is springs and streams without dense vegetation and mostly open shoreline by modeling the preferred micro- and macrohabitat of *Rana onca* and by conducting experimental manipulations at upper Blue Point Spring. Preliminary results indicate that *Rana onca* strongly avoided segments of high vegetation cover, especially where *Scirpus* spp. is present. *Rana onca* prefer areas with wider streams and areas with a higher percentage of shallow water. The vegetation preference of *Rana onca* appeared to be *Typha* spp. and *Eleocharis* spp.; *Rana onca* tended to avoid *Scirpus* spp. (Jaeger et al. 2004). A future study will statistically test *Rana onca* use of the experimental habitat manipulations that occurred in the previous study testing the hypothesis that reduction of vegetation cover and density at Blue Point Spring will increase the population of *Rana onca* present at the spring (Jaeger and Riddle 2005).

Movement

Rana onca is primarily nocturnal during the summer months and switches to a diurnal schedule during the winter. There is no evidence of torpor, hibernation, or migration for *Rana onca* (Center for Biological Diversity 2002). *Rana onca* is inactive in cold temperatures (AFGD 2003). *Rana onca* is restricted to narrow habitat corridors and will not move across desert habitats (Center for Biological Diversity 2002).

Rana onca as Distinct Taxon

Since its rediscovery, there have been questions whether populations of leopard frogs in the Black Canyon/Virgin River area are *Rana onca*, *Rana yavapaiensis*, or a hybrid of *Rana onca* and *Rana yavapaiensis*. Jaeger et al. (2001) used mitochondrial DNA and morphological analysis to determine that the seven populations of leopard frogs in the Black Canyon/Virgin River area were one distinct taxon (*Rana onca*) (Jaeger et al. 2001). Jaeger et al. (2001) recommended that conservation strategies that retain the leopard frogs in the Black Canyon/Virgin River (*Rana onca*) as evolutionary distinct units be developed (Jaeger et al. 2001). The systematic relationship between *Rana fisheri* (extinct Vegas Valley leopard frog) and *Rana onca* remains unresolved. Some authors believe that *Rana fisheri* and *Rana onca* are synonyms. Jennings et al. (1995) concluded that *Rana fisheri* is not synonymous with *Rana onca* and should be considered a valid, distinct species (Center for Biological Diversity 2002). The question of *Rana fisheri* systematic relationship to *Rana onca* is directly relevant to conservation efforts because many conservation actions may depend on a benefit from a clear understanding of the historical distributions (Center for Biological Diversity 2002).

Reasons for Decline and Threats

Rana onca has declined across its range and is vulnerable to extinction. The remaining habitat in which *Rana onca* occurs and its total population size is small. Dispersal among the remaining habitats has been reduced by the formation of Lake Mead (Bradford and Jennings 2005). Probable causes for the decline of *Rana onca* include loss or alteration of aquatic habitat for agriculture, urban and water development, degradation of habitat by cattle and wild burro grazing, emergent vegetation encroachment, and competition with non-native species (bullfrogs, non-native fish, crayfish, western spiny soft-shell turtle) (Bradford et al. 2004, Center for Biological Diversity 2002, Jennings 1988, Moyle 1973, Platz 1984). Water development has flooded historical *Rana onca* habitat, eliminated brief annual floods that would prevent the encroachment of emergent vegetation, and eliminated connectivity between remaining populations (Center for Biological Diversity

2002). Agriculture and urban development have destroyed *Rana onca* habitat (Center for Biological Diversity 2002). Extensive grazing can cause habitat degradation, although in some situations managed grazing of emergent vegetation may benefit *Rana onca* by providing open water habitats (Center for Biological Diversity 2002).

Threats to extant populations of *Rana onca* include non-native species, population fragmentation, small population size, low genetic variation, encroachment of emergent vegetation, right of way impacts, natural erosion, and recreational impacts (Center for Biological Diversity 2002, Natureserve 2005, RLFCT 2005). Hayes et al. (1986) stated that existing literature does not support the hypothesis held by many managers that American bullfrogs are the most important agent in ranid species decline, and other causes, such as non-native fish and habitat alteration, are equally responsible. Hayes et al. (1986) suggested that unless future studies prove that the American bullfrogs are the major cause for ranid species decline, uncontrollable density reductions are undesirable (Hayes et al. 1986). Non-native fish are recognized as a threat to *Rana onca* populations. Observations have been made that provide the basis for the hypothesis that nonnative fishes reduce eggs and tadpole survival, thus, limiting recruitment into adult populations at the Northshore Springs. A future study will determine whether introduced fish can be removed from a section of Blue Point Spring by use of fish barriers and common eradication techniques. The study will also test the hypothesis that eradication of non-native fish increases egg and tadpole presence and metamorph-juvenile frog recruitment from manipulated segments of the stream (Jaeger and Riddle 2005).

LCR MSCP Conservation Measures

RLFR1—Provide funding to support existing relict leopard frog conservation programs. LCR MSCP program activities will assist and contribute to existing relict leopard frog research and conservation programs where appropriate. In particular, the LCR MSCP will contribute $10,000 per year for 10 years to support implementation of planned, but unfunded, conservation measures for the relict leopard frog. To the extent consistent with the LCR MSCP Conservation Plan goals and objectives, implementation of this conservation measure will be coordinated with the Relict Leopard Frog Conservation Team.

The Relict Leopard Frog Team

The LCR MSCP calls for the program to contribute $10,000 per year for the next 10 years to conduct planned but unfunded conservation efforts. Current and future conservation efforts for *Rana onca* are coordinated under the Relict Leopard Frog Conservation Team (RLFCT), which include members of AFGD, the Bureau of Land Management (BLM), National Park Service (NPS), Nevada Department of Wildlife (NDOW), and the USFWS. Their conservation plan includes objectives for the next 10 years, a manual on *Rana onca* research, monitoring, translocation, and site-selection techniques, and work plans from 2003 to 2005 (RFLCT 2005).

Translocation

Translocation is a conservation measure that has been implemented by the RLFCT for *Rana onca*. Frogs were released at the Boulder City Wetlands in 2000 and 2001(RLFCT 2005, NPS). Eggs or tadpoles were collected from Bighorn Sheep Spring, Blue Point Spring, Rogers Spring, Boy Scout Spring and Saltcedar Spring (NPS). Eggs and tadpoles were reared in NPS and NDOW aquaria to metamorphosed frogs and then released in the wild (NPS). Four hundred and thirty seven juvenile frogs and tadpoles were released at the ponds (Ulepic 2002). Biologists limit collection to 5% of eggs and tadpoles at an individual site (NPS). Twenty one large adult *Rana onca* were observed at the ponds in November of 2001 (Ulepic 2002). Only one frog was observed at the wetland ponds in March of 2002, therefore; the site is no longer considered suitable habitat for *Rana onca* (Ulepic 2002, RLFCT 2005). Possible reasons for the low translocation success rate at the wetland ponds are presence of *Rana catesbeiana*, recreational development in surrounding area, and lack of habitat requirements that are critical to *Rana onca* (Ulepic 2002). One hundred and thirteen frogs and tadpoles were released in September of 2002 into Sugarloaf Spring. Additional potential release sites include: Nevada Hot Spring, Corral, Tassi, Grapevine, Pumphouse outflow, Pupfish, Sugarloaf and Lone Palm Springs. Members of the RLFCT will continue to survey other potential release sites (RLFCT 2005).

Current and Future Conservation Efforts by the Relict Leopard Frog Conservation Team (RLFCT 2005)

2003

- The Willow Beach Hatchery became a functioning frog-rearing facility.
- Grapevine Spring located in Arizona was evaluated and improved for translocation.
- Surveys of suitable translocation sites in *Rana onca* historical habitat were conducted.
- One hundred and sixty-four froglets were released into Sugarloaf Spring.
- Improvements were made to the frog-rearing facility at the National Park Service (NPS) by adding an external filtering system to all tadpole aquariums, and by adding artificial vegetation and more dry land to the frog tanks.
- One visual encounter survey was conducted at Blue Point Spring.
- Twenty-two frogs were detected at Blue Point Spring.
- Monitoring, site-selection criteria, and translocation protocol that is found in Appendix 5 of the conservation assessment and strategy were established.
- Pakoon, Hiko, and Gnatcatcher springs were found unsuitable for the translocation of *Rana onca*.
- Jaeger J.R., Bradford D.F., Jennings R.J., and Riddle B.R. presented *Conservation of the Relict Leopard Frog (Rana onca): Our Limited Understanding of the Distribution, Size, Structure, and Dynamics of Extant and Recently Extinct Populations* at the BIOS symposium, October 18, 2003. Department of Biological Sciences, University of Nevada, Las Vegas.

2004

- Improvements were made to the frog-rearing facility at Willow Beach Hatchery by installing a 20 ft by 2 ½ ft raceway and an 8 ft by 2 ½ ft tank.
- The Willow Beach Hatchery was explored for potential sites for the creation of a wetland area. Characteristics for suitable sites would be: 1) the area is similar to sites where *Rana onca* currently is found, 2) the area is protected from the public and other hatchery operations, and 3) the area has a good water source. A 100-m section of the hatchery was found to be suitable for the creation of the artificial habitat.
- One hundred and sixty-one froglets were released into Sugarloaf Spring, 391 froglets were released into the Pupfish Refuge Spring, 905 tadpoles were released into the Grapevine Spring, and 879 tadpoles were released into the Goldstrike Canyon Spring.
- Twenty-one spring sites on the Gold Butte grazing allotment were surveyed for suitable translocation sites; only two of these sites were found suitable and would require significant restoration before frogs could be translocated.
- Improvements were made to habitat at Pupfish Spring by clearing vegetative overgrowth in sections of the outflow stream. Several small rocks or sandbag dams were created in the cleared sections.
- Tassi and Redrock springs were evaluated and improved for translocation.
- All extant and recently introduced populations of *Rana onca* were monitored during the spring and fall.
    - Bighorn Sheep: Spring—188 adults, 10 juveniles, and 300 tadpoles and 54 egg masses; Fall—354 adults, 19 juveniles, and 69 tadpoles
    - Bluepoint: Spring—18 adults and 4 tadpoles; Fall—32 adults and 3 juveniles
    - Boy Scout: Spring—21 adults
    - Roger: Spring—5 adults; Fall—1 adult
    - Salt Cedar: Spring—4 adults, 3 juveniles, and 32 tadpoles
    - Goldstrike Canyon: Fall—15 adults
    - Sugarloaf: Spring—39 adults, 2 juveniles, and 22 tadpoles; Fall—32 adults and 3 tadpoles
    - Grapevine: Fall—6 adults
    - Pupfish Refuge Spring: Fall—18 adults
- The studies, *Temperature Acclimation and Oxygen Consumption of Rana onca Larvae,* and, *Evaluation of the Impact of Vegetation Encroachment on Relict Leopard Frog Population,* were initiated by the University of Nevada Las Vegas.
- Field coordination and site evaluation for potential relict leopard frog refugium sites in the upper Muddy River area were pursued with The Nature Conservancy (TNC), the Bureau of Land Management (BLM), and the U.S. Fish and Wildlife Service (USFWS).

2005

- Improvements will be made to the frog-rearing facility at Willow Beach Hatchery.
- Planning for the artificial habitat (warmwater well and outflow) at Willow Beach Hatchery will be implemented.
- The study, *Evaluation of the Impact of Vegetation Encroachment on Relict Leopard Frog Populations*, will be completed by the University of Nevada Las Vegas.
- The study, *Temperature Acclimation and Oxygen Consumption of Rana onca Larvae*, will be completed by the University of Nevada Las Vegas.
- Site evaluations will be conducted at Tassi Spring, Red Bluff Spring, Red Rock Spring, and a pond at Kingman's community college.
- Site maintenance will be conducted at Sugarloaf and Boy Scout Canyon springs.
- Visual encounter surveys will be conducted on all extant and recently introduced populations of *Rana onca*.
- Populations of *Rana onca* at Sugarloaf, Pupfish Refuge, Goldstrike Canyon, and Grapevine springs will be augmented.
- The habitat at Pupfish Refuge and Salt Cedar springs will be improved.
- A refugium at Ash Grove Spring and Muddy River will be developed.
- A GIS map of natural, transplanted, and potential sites will be developed.
- Tassi and Red Rock springs will be prepared to receive transplanted frogs.
- The following studies will be implemented: 1*)* relict leopard frog monitoring and management, 2) establishment of a *Rana onca* population in a created aquatic habitat, 3) delineation and distribution, evaluation of relatedness, and assessment of connectivity for leopard frog populations, and 4) evaluation of experimental habitat manipulations on relict leopard frog populations.

LCR MSCP Research and Monitoring Needs

LCR MSCP conservation measures are restricted to funding implementation of planned but unfunded conservation measures from existing conservation programs, through coordination with the Relict Leopard Frog Conservation Team. Additional research and monitoring will not be implemented by the LCR MSCP.

Other Research and Monitoring Opportunities

Specific conservation measures for *Rana onca* are coordinated by the relict leopard frog conservation team, but the following are general conservation needs cited in the current literature:

- Further research should be conducted on the life history of *Rana onca*, particularly feeding and breeding behavior (AGFD 2003, Natureserve 2005, RLFTC 2005).

- Locations in the historical range of *Rana onca* should continue to be surveyed. The southern limit of the *Rana onca* genotype downstream from Black Canyon is unknown. The tributaries of the Colorado River upstream of the confluence with the Virgin River have not been surveyed for *Rana onca* (Blomquist et al. 2003, Bradford et al. 2004).
- The systematic relationship of *Rana fisheri* and *Rana onca* should be determined (Bradford et al. 2004, Center for Biological Diversity 2002).
- Further research should be conducted on the habitat requirements of *Rana onca,* especially related to emergent vegetation encroachment (Natureserve 2005, Bradford et al. 2005). Current research previously mentioned has addressed some of these concerns.
- Further research on gene flow, genetic, and population structure is needed to provide a basis for managing genetic stock in the establishment of new populations. More structure on population connectivity of *Rana onca* is needed to provide insight on metapopulation structure (Jaeger et al. 2001, AFGD 2003). The current study, *Delineation of Distribution, Evaluation of Relatedness, and Assessment of Connectivity for Leopard Frog Populations (Rana spp.) Within the Management Zone of the Relict Leopard Frog (Rana onca),* conducted by the University of Nevada Las Vegas, seeks to answer some of these questions. The study had two objectives: 1) to assess the distribution of leopard frog populations within the Grand Canyon portion of the relict leopard frog Potential Management Zone, and determine the taxonomic identity of the leopard frog populations within this region, and 2) to identify of the rate of migration among extant populations of *Rana onca* and *Rana yavapiensis*, and to determine the amount of gene flow between *Rana yavapiensis* and *Rana onca* within the western Grand Canyon (Jaeger and Riddle 2005).
- Occupied habitat should be protected and eradicated of nonnative species (Natureserve 2005, RFLCT 2005, Blomquist 2003).
- The feasibility to use translocation to establish new populations in suitable unoccupied habitat should be assessed (Natureserve 2005, RLFCT 2005).
- Further research is needed on the effects of *Rana catesbiena* on *Rana onca*; current literature on this issue is lacking and management decisions are being based on the lack of sound scientific studies that link *Rana catesbiena* as a major factor in the decline of ranid species. Further research on this issue should include alternatives testable through manipulations, long-term demographic studies of populations at specific sites, and investigation of conditions that favor bullfrog survival (Hayes et al. 1986).
- Further research is needed on the ability of *Rana onca* to adapt to different temperatures for translocation purposes. The study, *Temperature Acclimation and Oxygen Consumption of Rana onca Larvae*, conducted by the University of Nevada Las Vegas, will attempt to answer this question. The objective of this study is to measure oxygen consumption of *Rana onca* larvae in different thermal environments to make an acute determination of temperature tolerance using thermal acclimation and stress level (Hoff and Haley 2004).

- If populations of *Rana onca* are found, a monitoring program of occupied and unoccupied habitats should be conducted to determine the status of *Rana onca* in the local area and determine the genetic relationship of these frogs to the nearest populations of *Rana onca* (Blomquist et al. 2003).
- Further research should be conducted on the importance of bighorn sheep, burros, and cattle in maintaining favorable habitat (Natureserve 2005).
- Illegal collection of *Rana onca* specimens and degradation of *Rana onca* habitat by the public should be prevented (RLFTC 2005).

Literature Cited

Arizona Game and Fish Department. 2003. Rana onca. Unpublished abstract compiled and edited by the Heritage Data Management System, Arizona Game and Fish Department, Phoenix, Arizona. 4 p.

Amphibia Web. 2005. Information on amphibian biology and conservation [web application]. Berkeley, California: AmphibiaWeb. Accessed on May 6, 2005. http://amphibiaweb.org/.

Blomquist, S.M., D.A Cox, and M.J. Sredl. 2003. Inventory and habitat assessment of the relict leopard frog (*Rana onca*) in Arizona. Nongame and Endangered Wildlife Program Technical Report 219. Arizona Game and Fish Department, Phoenix.

Bradford, D.F, J.R Jaeger, and R.D Jennings. 2004. Population status and distribution of a decimated amphibian, the relict leopard frog (*Rana Onca*). The Southwestern Naturalist 49:218-228.

Bradford, D.F., and R.D. Jennings. 2005. *Rana onca* Cope Relict Leopard Frog. *In:* Lanoo, M.J (editor), Status and Conservation of US Amphibians Volume 2: Species Accounts. University of California Press, Berkeley.

Center for Biological Diversity and Southern Utah Wilderness Alliance. 2002. Petition to list the relict leopard frog (*Rana onca)* as an endangered species under the endangered species act. Center for Biological Diversity, Berkeley, California, and Southern Utah Wilderness Alliance, Salt Lake City, Utah.

Figiel, C. 2005. Establishment of a *Rana onca* population in a created aquatic habitat. U.S. Fish and Wildlife Service. Clark County MSHCP Proposal. Last updated 5/04/2005.

Hayes, M.P., and M.R Jennings. 1986. Decline of ranid frog species in western North America: Are bullfrogs (*Rana catesbeiana*) responsible? Journal of Herpetology 20:490-509.

Hoff, and Haley. 2004. Temperature acclimation and oxygen consumption of *Rana onca* larvae. National Park Service. Clark County MSHCP Project. Last updated 4/15/2004.

Jaeger, J.R., B.R. Riddle, R.D. Jennings, and D.F. Bradford. 2001. Rediscovering *Rana onca*: Evidence for phylogenetically distinct leopard frogs from the border region of Nevada, Utah, and Arizona. Copeia 2001:339-354.

Jaeger, J., B. Riddle, and R. Haley. 2004. Evaluation of the impact of vegetation encroachment on relict leopard frog populations. University of Las Vegas

Nevada, and National Park Service. Clark County MSHCP project. Last updated 4/15/2004.

Jaeger, J., and B. Riddle. 2005. Evaluation of experimental habitat manipulations on Relict Leopard Frog Populations. University of Nevada Las Vegas. Clark County MSHCP Proposal. Last updated 05/04/2005.

Jaeger, J., and B. Riddle. 2005. Delineation of distribution, evaluation of relatedness, and assessment of connectivity for leopard frog populations (*Rana* spp.) within the management zone of the relict leopard frog (*Rana* onca). University of Nevada Las Vegas. Clark County MSHCP Proposal. Last updated 5/04/2005.

Jennings, M.R. 1988. *Rana onca* Cope, relict leopard frog. Catalogue of American Amphibians and Reptiles 417:1-2

Jennings, M.R., and M.P Hayes. 1994. Decline of native ranid frogs in the desert Southwest, p. 183-211. *In:* P.R. Brown and J.W Wright (editors), Herpetology of the North American deserts, proceedings of a symposium. Spec. Publ. 5. Southwestern Herpetologists Society. Serpent's Tale Books, Excelsior, Minnesota.

Malfatti, M. 1998. The relict leopard frog, *Rana onca.* Vivarium 9:36-37, 58-60.

Moyle, P.B. 1973. Effects of introduced bullfrogs, *Rana catesbeiana*, on the native frogs of the San Joaquin Valley, California. Copeia 1973:18-22.

National Park Service. Undated. Introduction of *Rana onca*, the relict leopard frog, into the Boulder City Wetlands Park. (Draft). National Park Service, Lake Mead Recreation Area. Boulder City, Nevada.

National Park Service. Undated. The relict leopard frog, *Rana onca*, at the bighorn sheep site. (Draft). National Park Service, Lake Mead Recreation Area. Boulder City, Nevada.

National Park Service. 2001. Relict leopard frog (*Rana onca*) historical synopsis. (Draft). National Park Service, Lake Mead Recreation Area. Boulder City, Nevada.

National Park Service. 2001. Roger's and Blue Point springs complex historical synopsis. (Draft). National Park Service, Lake Mead Recreation Area. Boulder City, Nevada.

Natureserve. 2005. NatureServe Explorer: An online encyclopedia of life [web application]. Version 4.4. NatureServe, Arlington, Virginia. Accessed on May 6, 2005). http:www.natureserve.org/explorer.

Platz, J.E. 1984. Status report for *Rana onca* Cope. Department of Biology, Creighton University. Prepared for Office of Endangered Species, U.S. Fish and Wildlife Service, Albuquerque, New Mexico.

Powell, E., and K. Turner. 2003. Floristic survey of select springs along the Colorado below Hoover Dam. Clark County MSHCP Proposal. National Park Service. Boulder City, Nevada.

Relict Leopard Frog Conservation Team. 2005. Conservation agreement and rangewide conservation assessment and strategy for the relict leopard frog (*Rana onca*).

Scanland, R., and J. Moore. 2003. Relict leopard frog recovery strategy—refugia proposal. Clark County MSHCP Project. The Nature Conservancy. Updated 4/15/2004.

Schwaibe, C.R., and P.C. Rosen. 1988. Preliminary report on the effect of bullfrogs in wetland herpetofaunas in southeastern Arizona. General Technical Report. U.S. Department of Agriculture, Forest Service. pp 166-173.

Ulepic, C.R. 2002. The introduction of *Rana onca* to the Boulder City Wetlands. (Draft). Senior Thesis. University of Nevada Las Vegas.

U.S. Fish and Wildlife Service. Federal Register Volume 69, No. 86, Tuesday, May 4, 2004. Retrieved from http://www.fws.gov/arizonaes.

FLANNELMOUTH SUCKER (*Catostomus latipinnis*)

Distribution

The flannelmouth sucker was historically the most abundant large fish species in the Upper Colorado River Basin (UCRB) (Vanicek et al. 1970, Holden 1973, Holden and Stalnaker 1975, McAda 1977). However, the flannelmouth sucker was found to be declining in the Lower Colorado River Basin (LCRB), and for a period it was largely restricted to the Colorado River above Lake Mead and a short segment of the Salt River (Minckley 1973). Flannelmouth sucker apparently are not able to persist in large impoundments, unlike the razorback sucker. Jonez and Sumner (1954) found razorback sucker to be relatively common in lakes Mead and Mohave, while flannelmouth sucker were only encountered rarely. This trend appears to continue, with annual catches of razorback sucker being the norm on Lake Mead, while only one flannelmouth sucker was captured during 9 years of intense field efforts associated with razorback sucker studies (BIO-WEST, Logan, UT, unpublished data), and only two flannelmouth sucker have been captured in Lake Havasu (Mueller and Marsh 2002). Interestingly, flannelmouth sucker may never have been vastly abundant in the LCRB. For example, Dill (1944) did not indicate the presence of flannelmouth sucker in the LCRB during his sampling efforts at the turn of the century. Miller (1961) reported few captures of the species the LCRB, but he stated that the species was found in the Salt and San Pedro rivers in the early 1900s. Furthermore, flannelmouth sucker was thought to be extirpated from the LCRB by the 1970s (Mueller and Wydoski 2004).

However, in 1976 the Arizona Game and Fish Department stocked 611 adult flannelmouth sucker originating from the Paria River into the mainstem of the LCRB, near Bullhead City, Arizona, for purposes of black fly abatement (Minckley 1979). Post-introduction flannelmouth sucker survival was uncertain, as Minckley (1979) reported that none of the fish were observed post-stocking. However, Mueller and Wydoski (2004) report that by the mid-1990s, young flannelmouth sucker were found by the Arizona Game and Fish Department and the Nevada Department of Wildlife. In 1998 Mueller and Wydoski initiated a study that documented natural recruitment in the highly altered 80 kilometer (km) reach of the Lower Colorado River downstream from Davis Dam. This introduction represents the first successful reintroduction of a native species in the highly altered LCRB (Mueller and Wydoski 2004). The successful reintroduction of flannelmouth sucker is of particular interest, because similar trends in flannelmouth sucker abundance and distribution were noted following disturbances from the construction of mainstem dams in the UCRB. For example, Wiltzius (1976) indicated that flannelmouth suckers were being replaced by introduced longnose suckers, a phenomenon likely attributable to the coldwater releases from mainstream dams and competitive interactions with nonnative species. However, flannelmouth sucker remain relatively abundant in the UCRB in less-altered sections of river (McAda 1977, McAda and Wydoski 1985, Tyus et al. 1982) and the most abundant large-bodied fish species in some river sections (Ryden 2005). As discussed below, the flannelmouth sucker is the generalist of the river, especially with regard to nursery habitat requirements, compared with the other rare species (razorback sucker, bonytail, pikeminnow, and other *Gila* species). The interesting suite of characteristics that have allowed for successful flannelmouth sucker re-establishment should be scrutinized in hopes of providing important insights applicable to establishing self-sustaining populations of this and other endemic Colorado River species.

Historical Habitat Modifications

Numerous researchers have identified that the major factor contributing to the decline of flannelmouth sucker and other large-river fishes has been the construction of mainstem dams and the resultant cool tailwaters and reservoir habitats that replaced a once warm, riverine environment (Holden and Stalnaker 1975, Joseph et al. 1977, Wick et al. 1982, Minckley et al. 1991). Competition and predation from nonnative fishes that are successfully established in the Colorado River and its reservoirs have also contributed to their decline (Minckley and Deacon 1991).

Furthermore, given declines of suitable riverine habitat, increased predation and competition in current suitable and limited habitats may occur (Mueller and Marsh 2002). For further detailed information including examples, ramifications, and research needs pertaining to the effects of habitat modifications on native Colorado River fishes, please see Tyus et al. (1982), Minckley and Deacon (1991), Chart and Bergersen (1992), and Mueller and Marsh (2002).

Systematics and Morphomethrics

The flannelmouth sucker belongs to the family Catostomidae. Likely the most conspicuous feature of this group is the distinct, fleshy lips ventrally located on the snout. Mouthparts of most catostomids are enlarged, protrusible, and plicate or covered with papillae, presumably adapted for benthic feeding strategies (Miller and Evans 1965). The genus *Catostomus* contains a large number of species, with several species from the Rocky Mountains; most of these western species are native and isolated to tributaries (Eddy and Underhill 1969). Flannelmouth sucker are distinguished from other species of the genus *Catostomus* by the thickened lower lip that is elongated compared with that other species, and completely divided by the median groove. Furthermore, dorsal ray counts are typically 10-11 and lateral line scales are generally more than 80. Scales located immediately above the lateral line tend to be bordered in dark pigment (Eddy and Underhill 1969, Mueller and Marsh 2002). Holden (1973) reports flannelmouth sucker as typically dark brownish-green dorsally, yellowish or orange laterally, and white ventrally. However, in more turbid reaches, flannelmouth sucker captured by Holden (1973) tended to be lighter shades of tan dorsally and silvery to white on the lateral and ventral surfaces. Colorations become more prominent during the spawning season, with tubercles becoming highly evident on male fish (Cross 1975, Muller and Marsh 2002). Adult flannelmouth sucker reach lengths over 18 in (Eddy and Underhill 1969), with some of the largest specimens being captured in the LCRB where lengths can exceed 26 in (Muller and Marsh 2002). Larval stages are best described in Snyder et al. (2004).

Hybridization

As reviewed by Bestgen (1990) and described above for razorback sucker, hybridization between flannelmouth sucker and other native Colorado River catostomid species has historically been documented to occur. Flannelmouth sucker have been shown to hybridize with populations of razorback sucker. It is foreseeable that populations of flannelmouth sucker would hybridize with Sonora sucker (*Catostomus insignis*) and other native catostomids, given that flannelmouth sucker presently exist in greater abundances in the Colorado River and its tributaries, as compared with other native fishes (Hubbs et al. 1943, Hubbs and Miller 1953, Holden 1973, Holden and Stalnaker 1975, McAda and Wydoski 1980, Minckley 1983, Bozek et al. 1984, Tyus and Karp 1990, Douglas and Marsh 1998). Buth et al. (1987) used allozymic data to directly quantify presumed introgression in the range of their samples as being 0-5% toward flannelmouth sucker and 0-3% toward razorback sucker. Hybridization between flannelmouth sucker and razorback sucker is problematic, given that adults of both species have been documented to simultaneously utilize cobble-bottomed, main-channel riffles for spawning activities in natural riverine settings (see razorback sucker profile, this report). A larger concern currently is hybridization between flannelmouth sucker and introduced sucker species. Hybridization has been documented between flannelmouth sucker and white sucker in most UCRB streams (Holden 1973, Ryden 2005).

Habitat

Adults

Historically, flannelmouth sucker inhabited virtually all components of riverine habitat ranging from fast current, to riffle, eddy, and stagnant backwater areas. Flannelmouth sucker have been known to be the predominate species comprising native fish catches (Holden 1973, Holden and Stalnaker 1975, McAda 1977, Holden 1999). Minckley (1973) described the flannelmouth sucker as being an inhabitant of the larger, swifter streams and rivers of the Colorado River Basin. Cross (1975) found flannelmouth sucker to occur 64% of the time in runs, 31% in pools, and only occasionally in riffle habitat types in the Virgin River. Furthermore, Cross (1975) documents that while flannelmouth sucker were collected over all types of substrate, ranging from mud-bottomed flats to boulders, the majority of collections were made when sampling sand and rubble-cobble substrates (60% and 35%, respectively). Flannelmouth sucker also appear to have affinity for overhead cover, with more than 50% of the collections associated with boulders, overhanging trees, or undercut banks in the relatively small Virgin River (Cross 1975). Flannelmouth sucker were typically found in deeper water (mean 59 cm ± 49 cm in the Virgin River) and water velocities at capture were variable (0-1.0 m/s, mean value of 0.44 m/s) (Cross 1975). More applicable to mainstem Colorado River habitats, Gaufin et al. (1960) found adult flannelmouth sucker in the Green River to be most abundant in slower, deeper sections, similar to those described by Cross (1975). McAda and Wydoski (1980) report that flannelmouth sucker were most often captured at the lower portions of a glide or pool and in the vicinity of a mud-silt bottom. Mueller and Wydowski (2004) report flannelmouth sucker captures typically in water ranging to 2 m deep in the lower Colorado River. More rigorous studies of fairly long river reaches have shown that flannelmouth sucker populations are larger in reaches with large amounts of cobble-gravel substrates and smaller in areas of predominately sand substrate (Ryden 2005), just the opposite of razorback sucker.

Chart and Bergersen (1992) documented the migrational impacts on flannelmouth sucker as a result of a mainstream impoundment on the White River, both pre- and post-construction of the dam on the White River, Colorado. Recapture data suggest that the dam blocked the return of adult flannelmouth sucker to their home ranges after dam closure, indicating the likelihood of large, seasonal, migrational movements historically. Furthermore, Vanicek et al.(1970) found flannnelmouth sucker and bluehead sucker (*Catostomus discobolus*) to be the only native species to persist below Flaming Gorge Dam. This finding is likely attributable to the cobble-bottomed, riffle habitats typically created below large impoundments, a known habitat important to flannelmouth sucker (Holden 1999). Radio telemetry work by Beyers et al. (2001) suggests that habitat use by adult flannelmouth sucker does not change with time of day, unlike other native fishes that tend to display unique diel habitat use patterns (see bonytail species profile).

Furthermore, flannelmouth sucker tracked by radio telemetry were found in water ranging from 0.5 m to greater than 3.0 m depth, with the most contacts made in depths of 1.5 m. Cross (1975) mentions that flannelmouth sucker were collected the majority of time in “unmodified” physical habitat (80% of collections) with “occasional” collections in habitats of “poor” water quality. Finally, McAda (1977) suggests that the flannelmouth sucker is not as specific in its habitat selection, compared with other large-bodied native fishes (e.g., razorback sucker, bonytail, humpback chub [*Gila cypha*], and Colorado pikeminnow [*Ptychocheilus lucius*]). However, unlike razorback sucker that continue to persist in reservoir situations, flannelmouth sucker apparently either do not survive well in reservoirs, or avoid large, lentic habitats, and therefore, exhibit at least some degree of an obligatory riverine life history strategy (Mueller and Marsh 2002).

Flannelmouth sucker have been commonly captured in water temperatures ranging from 10 to 35°C (mean of 24°C, conductivities varied from 150 to 2,700 mmhos) (Cross 1975). Deacon et al. (1987) found the final thermal preferendum of flannelmouth sucker to be 25.9°C (±0.5°C) using laboratory techniques, but they suggest that the upper temperature threshold determining habitat usage is highly dependant upon acclimation temperature and that flannelmouth sucker, in particular, have one of the highest temperature tolerances displayed by native fishes of the Colorado River. Ward et al. (2002) tested the effects of temperature, fish length, and exercise on the swimming performance of young-of-year flannelmouth sucker. Results suggest that fatigue velocities increased with fish size and water temperature, suggesting that warmer in-river temperatures may be important for flannelmouth sucker recruitment and survival. This hints at the relatively high thermal preferences of flannelmouth sucker and the potential impacts that coldwater releases may have on populations of this species. Carter and

Hubert (1995) found the upper elevational threshold for flannelmouth sucker inhabitance to be 2,192 m based on studies conducted in southwestern Wyoming.

Spawning

Flannelmouth sucker initiate spawning during May and June in the UCRB, when water temperatures are between 6-12°C (McAda 1977, Weiss et al. 1998). Ripe male fish have been captured through July; however, ripe females are rarely collected after the first few weeks in June (Holden 1973, McAda 1977). In contrast, Mueller and Marsh (2002) report spawning to occur in April and May in the LCRB. Historically, flannelmouth sucker, as well as other native catostomids, were reported to make impressive spawning migrations (Simon 1946, La Rivers 1962). More recently, relatively little migrational movement has been observed, presumably due to the impacts associated with mainstem impoundments (Holden 1973, McAda 1977). McAda (1977) captured ripe male and female razorback sucker over cobble and gravel bars in the Yampa and Colorado rivers during spawning season. This is corroborated by research on the San Juan River, as adults have been found spawning over in-channel, cobble-bottomed riffles (Holden 1999). Most recently in the LCRB, spawning activities of aggregations of greater than 200 flannelmouth sucker were visually observed and sampled below Davis Dam to Lake Havasu. Spawning fish were found congregating in water approximately 1-2 m deep, over large cobble and gravel substrates, and in relatively swift currents (0.5-1.0 m/s) (Mueller and Wydoski 2004). To the best of our knowledge, flannelmouth sucker do not spawn in reservoir or other still-water situations, and reproduction is largely limited to riverine settings, particularly main-channel riffles.

Larvae

As mentioned for razorback suckers, dispersal of larval flannelmouth sucker is also important and heavily influenced by flow regimes (Robinson et al. 1998). However, unlike razorback sucker larvae, young flannelmouth sucker apparently do not exhibit diel drift periodicity. They do appear to be associated with near-shore (defined as shoreline habitats with velocities <0.2 m/s), slackwater rearing habitat types, indicating at least some degree of active affinity for drift habits (Robinson et al. 1998). Larval flannelmouth suckers are found in shoreline backwaters, embayments, and other low-velocity habitats in the San Juan River using larval seines (Brandenburg et al. 2005).

Juveniles

Juvenile flannelmouth sucker move out of backwaters and embayments 2-3 months after hatching. This is generally indicated by their sudden disappearance from seine collections, which concentrate on these low-velocity habitats. Juveniles tend to move into habitats with more velocity such as runs and edges of riffles (Holden 1999). This is very similar to habitat shifts that occur with roundtail (*Gila robusta*) and humpback chub (Holden 1977). This habitat shift appears to coincide with a decline in numbers, but this may be an artifact of poor sampling of these habitats. Lastly, Gido et al. (1997) suggest that secondary channels are important to young flannelmouth sucker in the San Juan River, but they do not compare use of those habitats with use of similar habitats in the main channel. Therefore, whether the species uses secondary channels or the habitats found in secondary channels is unclear.

Reproduction

Male and female flannelmouth sucker mature between 4 and 6 years of age in the UCRB, or at lengths ranging from 391 to 421 mm (McAda 1977, McAda and Wydoski 1985). Fecundity of the flannelmouth sucker is highly dependant on size, and is therefore highly variable within age groups (see age and growth section for reasoning). Female flannelmouth sucker produce anywhere from 3,000 to 30,000 eggs, depending on age and size. Furthermore, mature individuals can reproduce for more than 20 years, demonstrating remarkable reproductive abilities that help establish and maintain flannelmouth sucker populations in areas where other native species have declined and continue to decline (Mueller and Wydoski 2004). Even within year classes, numbers of individual eggs produced can differ by 20,000 eggs in some cases (McAda 1977). Sex ratios of male:female fish have been reported as 3:1 (McKinney et al. 1999) and 1:1-2:1 depending on capture location and time of year (Weiss et al. 1998). Egg diameters range from 2.5 to 3.8 mm, and eggs are typically deposited in distinct areas. After depositing eggs, females typically move on to new spawning areas, while males remain and await the arrival of another female (Weiss et al. 1998).

Diet

Flannelmouth sucker diet composition has been described as being omnivorous, with a wide variety of consumption ranging from algae to detritus, including mud and invertebrates (Ellis 1914, Sigler and Miller 1963, Taba et al. 1965, Minckley 1973, Cross 1975). More specifically, Cross (1975) described specimens from the Virgin River as consuming aquatic insect larvae (*Diptera*, *Ephemeroptera*, *Tricoptera*, and even amphipods), with portions of inorganic and organic material including quantities of filamentous algae. These findings are corroborated by efforts in the UCRB, as larval and early juvenile flannelmouth sucker were discovered to consume chironomids in greatest abundance, with evidence of organic and inorganic matter, diatoms, and unidentified plant seeds identified in the stomach contents.

Bartschi (1964) (as described by McAda [1977]) documented seasonal and size-specific shifts in flannelmouth sucker food habits. For example, flannelmouth sucker less than 80 mm in length fed exclusively on copepods, while fish greater than 200 mm in length did not consume copepods. As flannelmouth sucker increase in size, it appears that Ephemeroptera instars become an even more important food item, while other aquatic invertebrates (e.g., Diptera) are eaten by all size classes of flannelmouth sucker, particularly in the later spring and summer months. Furthermore, Ephemeroptera nymphs increase in importance during late summer into fall. Overall, the food habits of the flannelmouth sucker are highly dependant upon the availability of food items, with more common items becoming more predominant in diet composition at any given time. This strategy is typical of an omnivorous, opportunistic riverine obligate species (McAda 1977).

Age and Growth

Published growth estimates for flannelmouth sucker vary, and available information is highly dependant on age and habitat occupied (for details, see age and growth tables compiled by McAda [1977]). McAda (1977) used scales as a method for age determination and for back calculation of growth of UCRB flannelmouth sucker. Annuli formation was found to occur in June and July, with scales forming on young flannelmouth sucker as small as 25 mm. Scale development was complete throughout the surface of young fish by 40 mm. Total lengths of older fish were found to be variable, and considerable overlap in size was noted between age groups. Overall, age-I fish were found to be between 57 and 80 mm, age-II between 128 and 167 mm, age-III between 245 and 286 mm, age-IV between 351 and 370 mm, age-V between 412 and 432 mm, age-VI between 440 and 475 mm, age-VII between 446 and 493, and age-VIII between 456 and 514 mm (McAda 1977). McKinney et al. (1999) report growth for flannelmouth sucker collected below Lee’s Ferry, with fish displaying nearly isometric growth. Adults typically grew 5.5 mm/year (standard deviation 1.9 mm) and subadults 45.9 mm/year (standard deviation 16.8 mm) based on recaptured, tagged individuals. More recently, Mueller and Wydoski (2004) found that larger aging structures (such as entire fin rays and vertebrae) produced more discernable age information compared with otoliths and smaller fin ray sections. Length distribution data compiled by Mueller and Wydoski (2004) suggests that recruitment of flannelmouth sucker to the spawning cohort documented in the LCRB ranges between 9.4 and 31.3% per year, suggesting the capability of flannelmouth sucker to maintain a population of spawning-aged fish despite extensive riverine habitat modifications.

Literature Cited

Bartschi, D. 1964. A flannelmouth sucker stomach analysis. Unpublished manuscript. Utah Cooperative Fisheries Research Unit, Utah State University, Logan.

Bestgen, K.R. 1990. Status review of the razorback sucker, *Xyrauchen texanus*. Submitted to Bureau of Reclamation. Contribution 44, Larval Fish Laboratory, Colorado State University, Ft. Collins.

Beyers, D.W., C. Sodergren, J.M. Bundy, and K.R. Bestgen. 2001. Habitat use and movement of bluehead sucker, flannelmouth sucker, and roundtail chub in the Colorado River. Final Report to Colorado Division of Wildlife. Contribution 21, Larval Fish Laboratory, Department of Fishery and Wildlife Biology, Colorado State University, Fort Collins. 30 p.

Boyd, C.E. 1979. Water quality in warmwater fish ponds. Agricultural Experiment Station, Auburn University, Auburn, Alabama.

Bozek, M.A., L.J. Paulson, and J.E. Deacon. 1984. Factors affecting reproductive success of bonytail chubs and razorback suckers in Lake Mohave. Final Report. Bureau of Reclamation, Boulder City, Nevada.

Brandenburg, W.H., M.A. Farrington, and S.J. Gottlieb. 2005. Colorado pikeminnow and razorback sucker larval fish survey in the San Juan River during 2004. San Juan River Basin Recovery Implementation Program, U.S. Fish and Wildlife Service, Albuquerque, New Mexico.

Buth, D.G., R.W. Murphy, and L. Ulmer. 1987. Population differentiation and introgressive hybridization of the flannelmouth sucker and of hatchery and native stocks of the razorback sucker. Transactions of the American Fisheries Society 116:103-110.

Carter, B., and W.A. Hubert. 1995. Factors influencing fish assemblages of a high-elevation desert stream system in Wyoming. Great Basin Naturalist 55:169-173.

Chart, T.E., and E.P. Bergersen. 1992. Impact of mainstream impoundment on the distribution and movements of the resident flannelmouth sucker (Catostomidae: *Catostomus latipinnis*) population in the White River, Colorado. Southwestern Naturalist 37:9-15.

Cross, J.N. 1975. Ecological distribution of the fishes of the Virgin River. M.S. Thesis, University of Nevada, Las Vegas. 187 p.

Deacon, J.E., P.B. Schumann, and E.L. Stuenkel. 1987. Thermal tolerances and preferences of fishes of the Virgin River System (Utah, Arizona, Nevada). Great Basin Naturalist 47: 538-545.

Dill, W.A. 1944. The fishery of the lower Colorado River. California Fish and Game 30:109-211.

Douglas, M.E., and P.C. Marsh. 1998. Population and survival estimates of *Catostomus latipinnis* in Northern Grand Canyon, with distribution and abundance of hybrids with *Xyrauchen texanus*. Copeia 1998:915-925.

Eddy, S., and J.C. Underhill. 1969. How to know the freshwater fishes. 3rd edition. WCB/McGraw-Hill, Boston, Massachusetts.

Ellis, M.M. 1914. Fishes of Colorado. University of Colorado Studies 11:1-136.

Gaufin, A.R., G.R. Smith, and P. Dotson. 1960. Aquatic survey of Green River and tributaries within the Flaming Gorge Reservoir basin. University of Utah Anthropology Papers No. 48.

Gido, K.B., D.L. Propst, and M.C. Molles Jr. 1997. Spatial and temporal variation of fish communities in secondary channels of the San Juan River, New Mexico and Utah. Environmental Biology of Fishes 49:417-434.

Holden, P.B. 1973. Distribution, abundance and life history of the fishes of the Upper Colorado River Basin. Ph.D. Dissertation. Utah State University, Logan.

Holden, P.B. 1977. A study of the habitat use and movement of the rare fishes in the Green River from Jensen to Green River, Utah, August and September, 1977. Final Report for Western Energy and Land-use Team, Contract 14-16-0009-77-050, BIO-WEST, Logan, Utah.

Holden, P.B. (editor). 1999. Flow recommendations for the San Juan River. San Juan River Basin Recovery Implementation Program, U.S. Fish and Wildlife Service, Albuquerque, New Mexico.

Holden, P.B., and C.B. Stalnaker. 1975. Distribution of fishes in the Dolores and Yampa River systems of the upper Colorado Basin. Southwestern Naturalist 19:403-412.

Hubbs, C.L., L.C. Hubbs, and R.E. Johnson. 1943. Hybridization in nature between species of catostomid fishes. Contributions of the Laboratory of Vertebrate Biology, University of Michigan 22:1-76.

Hubbs, C.L., and R.R. Miller. 1953. Hybridization in nature between the fish genera *Catostomus* and *Xyrauchen*. Papers of the Michigan Academy of Sciences, Arts, and Letters 38:207-233.

Jonez, A., and R.C. Sumner. 1954. Lakes Mead and Mojave investigations: A comparative study of an established reservoir as related to a newly created impoundment. Federal Aid to

Fisheries Restoration Project Completion Report, F-1-R, Nevada Fish and Game Commission, Reno. 186 p.

Joseph, T.W., J.A. Sinning, R.J. Behnke, and P.B. Holden. 1977. An evaluation of the status, life history, and habitat requirements of endangered and threatened fishes of the Upper Colorado River Basin. Report FWS/OBS-77/2, U.S. Fish and Wildlife Service, Fort Collins, Colorado.

LaRivers, I. 1962. Fishes and fisheries of Nevada. Nevada Game and Fish Commission, Carson City.

McAda, C.W. 1977. Aspects of the life history of three catostomids native to the upper Colorado River basin. M.S. Thesis. Utah State University, Logan. 116 p.

McAda, C.W., and R.S. Wydoski. 1980. The razorback sucker, *Xyrauchen texanus*, in the upper Colorado River basin, 1974-76. U.S. Fish and Wildlife Service Technical Paper 99:1-15.

McAda, C.W., and R.S. Wydoski. 1985. Growth and reproduction of the flannelmouth sucker, *Catostomus latipinnis* in the upper Colorado River Basin 1975-76. Great Basin Naturalist 45:281-286.

McKinney, T., W.R. Persons, and R.S. Rogers. 1999. Ecology of flannelmouth sucker in the Lee's Ferry tailwater, Colorado River, Arizona. Great Basin Naturalist 59:259-265.

Miller, R.R. 1961. Man and the changing fish fauna of the American Southwest. Papers of the Michigan Academy of Science, Arts, and Letters. 46:365-404.

Miller, R.J., and H.E. Evans. 1965. External morphology of the brain and lips in catostomid fishes. Copeia 1965:467-487.

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

Minckley, W.L. 1979. Aquatic habitats and fishes of the lower Colorado River, Southwestern United States. Final Report to Bureau of Reclamation, Lower Colorado Region. Contract No. 14-06-300-2529. 478 p.

Minckley, W.L. 1983. Status of the razorback sucker, *Xyrauchen texanus* (Abbott), in the lower Colorado River basin. Southwestern Naturalist 28:165-187.

Minckley, W.L., and J.E. Deacon. 1991. Battle against extinction: Native fish management in the American West. University of Arizona Press. Tucson. 517 p.

Minckley, W.L., P.C. Marsh, J.E. Brooks, J.E. Johnson, and B.L. Jensen. 1991. Management toward recovery of razorback sucker. Pages 303–357 *In:* W.L. Minckley and J.E. Deacon (editors). Battle against extinction: Native fish management in the American Southwest. University of Arizona Press. Tucson. 517 p.

Mueller, G.A., and P.C. Marsh. 2002. Lost, a desert and its native fishes: A historical perspective of the lower Colorado River. Information and Technology Report USGS/BRD/ITR—2002-0010. U.S. Government Printing Office, Denver, Colorado, 69 p.

Mueller, G.A., and R. Wydoski. 2004. Reintroduction of the flannelmouth sucker in the lower Colorado River. North American Journal of Fisheries Management 24:41-46.

Robinson, A.T., P.P. Hines, J.A. Sorensen, and S.D. Bryan. 1998. Parasites and fish health in a desert stream, and management implications for two endangered species. North American Journal of Fisheries Management 18:599-608.

Robinson, A.T., R.W. Clarkson, and R.E. Forrest. 1998. Dispersal of larval fishes in a regulated river tributary. Transactions of the American Fisheries Society 127:772-786.

Ryden, D.W. 2005. Long-term monitoring of sub-adult and adult large-bodied fishes in the San Juan River: 2004. San Juan River Basin Recovery Implementation Program, U.S. Fish and Wildlife Service, Albuquerque, New Mexico.

Sigler, W.F., and R.R. Miller. 1963. Fishes of Utah. Utah Department of Fish and Game, Salt Lake City, Utah.

Simon, J.R. 1946. Wyoming fishes. Bulletin No. 4. Wyoming Game and Fish Department, Cheyenne. 129 p.

Snyder, D.E., R.T. Muth, and C.L. Bjork. 2004. Catostomid fish larvae and early juveniles of the upper Colorado River basin-morphological descriptions, comparisons, and computer-interactive key. Contribution 139 of the Larval Fish Laboratory, Colorado State University. Technical Publication No. 42, Colorado Division of Wildlife, Fort Collins, Colorado.

Taba, S.S., J.R. Murphy, and H.H. Frost. 1965. Notes on the fishes of the Colorado River near Moab, Utah. Proceedings of the Utah Academy of Science, Arts and Letters 42:280-283.

Tyus, H.M., B.D. Burdick, R.A. Valdez, C.M. Haynes, T.A. Lytle, and C.R. Berry. 1982. Fishes of the upper Colorado River Basin, distribution, abundance, and status. Pages 12-70 *In:* Miller, W.H., H.M. Tyus, and C.A. Carlson (editors). Fishes of the upper Colorado River System: Present and future. American Fisheries Society, Bethesda, Maryland. 131 p.

Tyus, H.M., and C.A. Karp. 1990. Spawning and movements of the razorback sucker *Xyrauchen texanus* (Abbott) in the Green and Yampa river, Colorado and Utah. Southwestern Naturalist 35:427-433.

Vanicek, C.D., R.H. Kramer, and D.R. Franklin. 1970. Distribution of Green River fishes in Utah and Colorado following closure of Flaming Gorge Dam. Southwestern Naturalist 14:297-315.

Ward, D.L., O.E. Maughan, and S.A. Bonar. 2002. Effects of temperature, fish length, and exercise on swimming performance of age-0 flannelmouth sucker. Transactions of the American Fisheries Society 131:492-497.

Weiss, S.J., E.O. Otis, and O.E. Maughan. 1998. Spawning ecology of flannelmouth sucker, *Catostomus latipinnis* (*Catostomidae*), in two small tributaries of the lower Colorado River. Environmental Biology of Fishes 52:419-433.

Wick, E.J., C.W. McAda, and R.V. Bulkley. 1982. Life history and prospects for recovery of the razorback sucker. Pages 120-126 *In:* W.H. Miller, H.M. Tyus, and C.A. Carlson (editors). Fishes of the upper Colorado River system: Present and future. American Fisheries Society, Bethesda, Maryland.

Wiltzius, W.J. 1976. Some historic influences of reservoirs and irrigation diversions on flows, temperatures, and fish distribution in the Gunnison River. Colorado Division of Wildlife, Fort Collins, Colorado. 100 p.

MACNEILL'S SOOTYWING SKIPPER (*Hesperopsis gracielae* MacNeill)

Legal Status

MacNeill's sootywing skipper (*Hesperopsis gracielae* MacNeill) was previously a Federal category 2 candidate and is currently listed as a species of concern. In Nevada, it is listed as S1 (critically imperiled) (Nevada Natural Heritage Program 2000). In California, this skipper is listed as S1 (extremely endangered) and S3 (restricted range, rare) (California Fish and Game Department 2002). In Arizona, it is currently listed as S? (uncertain) (Arizona Game and Fish Department 2003).

Distribution

Historical Range

MacNeill's sootywing skipper historical range was along the lower Colorado River and near the river along its tributaries in southeastern California, western Arizona, southern Nevada, and southern Utah (Scott 1986). Its type locality is along the California side of the lower Colorado River, near the Parker Dam, Arizona.

Current Range

The current range of the MacNeill's sootywing skipper approximates the historical ranges, except its presence in southern Utah is uncertain.

Populations within LCR MSCP Project Boundary

MacNeill's sootywing skipper populations have been detected during sweep-net collections of butterflies along the lower Colorado River (Nelson and Andersen 1999). Known populations have been observed in the following areas:

- Cibola National NWR (including 1993-1994 Unit B revegetation sites, and 1990-1991 Island Unit revegetation site)
- Havasu NWR (1993 Lost Lake revegetation site)
- near Cibola NWR (1978 Dredge Spoil revegetation site)
- Bill Williams River NWR (near Lake Havasu)
- Colorado River Indian Reservation (1987 No-Name Lake revegetation site)

Life History

General Description
MacNeill's sootywing skipper is a small (wingspread 23 mm) skipper with dark-brown and black mottled wings (MacNeill 1970). Skippers are butterflies with widely-spaced antennae that are usually hooked.

Life Cycle
The MacNeill's sootywing skipper deposits white or red spherical eggs singly on quailbrush (*Atriplex lentiformis*) leaves. Larvae may undergo two instars as it matures. Each mature larva cuts and wraps a leaf around itself, securing it with silk produced near the mouth. The insect pupates within the wrapped leaf and likely overwinters as pupae. Two or three adult flights occur per year (MacNeill 1970).

Diet
Larvae feed only on quailbrush. Adults require nectar, which is not provided by quailbrush.

Habitat
MacNeill's sootywing skipper requires dense stands of quailbrush, mixed with nectar-producing plants (specificity unknown). Species also may require quailbrush with high leaf water content resulting from shallow groundwater.

Threats
The species was originally listed as Federal category 2 candidate due to conversion of riparian habitat to agriculture in Moapa Valley, Nevada. The greatest historical cause of species decline likely was xerification of habitat due to river channelization and lowered water tables. The greatest present threat likely is destruction of habitat for housing development. An additional threat may be replacement of native quailbrush with cultivated varieties.

LCR MSCP Conservation Measures

MNSW1—Conduct surveys and research to locate MacNeill's sootywing skipper habitat and to better define its habitat requirements.
Research is being conducted to locate MacNeill's sootywing skipper populations that could be affected by covered activities and determine the macrohabitat and microhabitat requirements and ecology of the species. Based on research results, adaptive management experiments will be implemented to develop habitat establishment and management methods.

MNSW2—Create at least 222 acres of MacNeill's sootywing skipper habitat.
Based on results of research conducted under conservation measure MNSW1, at least 222 acres of MacNeill's sootywing skipper habitat will be created in reaches 1-4 near occupied habitat. Patches of created habitat will be designed and managed to support a mix of honey mesquite type III and quailbush to provide food plants for caterpillars and adults and to maintain the microhabitat conditions required by the species. A substantial amount of the 1,320 acres of honey mesquite type III that would be created is expected to be created in reaches occupied by

this species and will be established in conjunction with quailbush, the species' larval host plant. Consequently, it is anticipated that substantially more than 222 acres of habitat could be created under the LCR MSCP.

LCR MSCP Research and Monitoring Needs

Surveys are needed on the insect and its host plant within LCR MSCP boundaries (historical floodplain of LCR from upstream end of Lake Mead to SIB). Surveys should concentrate on river and wash inflows to the LCR (e.g., Virgin, Muddy, and Bill Williams rivers and Sacramento Wash). Surveys should record GPS coordinates of stands of *A. lentiformis* and estimate the plant's area. *Hesperopsis gracielae* should be detected as eggs, larvae, pupae, or adults on host plants and as adults on nearby nectar sources. Surveys should be conducted during April to October when adults are intermittently present (2-3 generations occur per season) and be completed within 2 years (2006 and 2007). *Hesperopsis gracielae* that are found should be digitally photographed and their GPS coordinates recorded. Densities (individuals of each life stage per plant or plant area (m^2) should be estimated.

Measure site factors affecting presence or absence of sootywing concurrent with surveys. Possible site factors are:

a. soil moisture
b. soil salinity
c. soil nitrogen
d. plant water content (estimated by weighing, drying, and reweighing branches)
e. availability of nearby nectar sources (distances, amounts, species)
f. area of *A. lentiformis* stand
g. plant genome (native plant or USDA-NRCS revegetation variety)
h. elevation
i. latitude

Literature Cited

Arizona Game and Fish Department. 2003. Heritage Data Management System: MacNeill's sooty wing skipper abstract. Available at http://www.azgfd.gov.

California Department of Fish and Game. 2002. State and Federally Listed Endangered, Threatened, and Rare Plants of California.

MacNeill, C.D. 1970. A new *Pholisora* with notes on *P. Alpheus* (Edw.) (Lepidoptera: Hesperiidae). Entomological News 81:177-184.

Nelson, S.M., and D.C. Andersen. 1999. Butterfly (Papilionoidea and Hesperioidea) assemblages associated with natural, exotic, and restored riparian habitats along the lower Colorado River, USA. Regulated Rivers: Research & Management 15:485-504.

Nevada Natural History Program. 2000. List of Sensitive Animals.

Scott, J.A. 1986. The butterflies of North America. Stanford University Press.

STICKY BUCKWHEAT
(*Eriogonum viscidulum* Howell)

Legal Status

The sticky buckwheat is Federally listed as a species of special concern. In Nevada, it is listed as fully protected by the Nevada Heritage Program.

Distribution

Historical Range

The historic range of the sticky buckwheat was restricted to the Muddy River and possibly the lower Virgin River (Powell, 2003).

Current range

The current range is restricted to Clark County, Nevada, along the Muddy River, the lower Virgin River (possibly into Mohave County, Arizona), and the Overton Arm of Lake Mead (Nevada Heritage Program 2001a).

Populations within LCR MSCP Project Boundary

The sticky buckwheat is present, within the LCR MSCP project boundary, near the Overton Arm of Lake Mead (Powell 2003, Bangle 2005).

Life History

General Description

The sticky buckwheat is an annual herb in the buckwheat family (Polygonaceae). Sticky buckwheat has branched stems, rising 4 dm in height above a basal rosette of leaves. Stems are sticky with adhering sand particles. Flowers are small and yellow (Nevada Heritage Program 2001b).

Phenology

Sticky buckwheat flowers from April through June (Nevada Heritage Program 2001b).

Habitat

This plant mainly grows in sandy soils (Nevada Heritage Program 2001b).

Threats

Major threats are rising reservoir elevations and competition from nonnative (saltcedar and Sahara mustard) and native (arrowweed) plants. Minor threats are trampling or browsing by burros. Trampling by people is rare (Powell 1999, Powell 2003, Bangle 2005).

LCR MSCP Conservation Measures

STBU1—Provide funding to support existing sticky buckwheat conservation programs.
The LCR MSCP will provide $10,000 per year until 2030 to the Clark County MSHCP Rare Plant Workgroup to support implementation of conservation measures for the sticky buckwheat and threecorner milkvetch that are beyond the permit requirements of the Clark County MSHCP.

LCR MSCP Research and Monitoring Needs

LCR MSCP conservation measures are restricted to providing funding to support existing sticky buckwheat conservation programs. Research and monitoring will not be conducted through LCR MSCP implementation.

Other Research and Monitoring Opportunities

The National Park Service has identified several research and monitoring needs, including: 1) effects of nonnative plants (red brome, Mediterranean grass, tumbleweed, and Tournefort's birdrape), 2) requirement for periodic flooding, 3) additional surveys, and 4) ecological studies (Powell 1999, Powell 2003).

Literature Cited

Bangle, D. 2005. Report on *Eriogonum viscidulum* (sticky buckwheat) monitoring (1997-2005) within Lake Mead National Recreation Area. National Park Service. Unpublished report.

Nevada Natural Heritage Program. 2001a. Nevada Rare Plant Atlas. Available at http://heritage.nv.gov/atlas/maps.

Nevada Natural Heritage Program. 2001b. Rare Plant Fact Sheet. Available at http://heritage.nv.gov/atlas/maps

Powell, E. 1999. Report on 1997 surveys for sticky buckwheat, *Eriogonum viscidulum*, in Lake Mead National Recreation Area. National Park Service. Unpublished report.

Powell, E. 2003. Report on sticky buckwheat (*Erigonum viscidulum*) monitoring, 2003, Lake Mead National Recreation Area. National Park Service. Unpublished report.

THREECORNER MILKVETCH
(*Astragalus geyeri* var. *triquetrus* [Gray] Jones)

Legal Status

The threecorner milkvetch was previously listed as a Federal category 2 candidate. Currently, it is listed as a species of concern. In Nevada, it is listed as critically endangered.

Distribution

Historical Range
Historically the threecorner milkvetch was found in southeastern Nevada and northwestern Arizona (Powell 1998).

Current Range
In Nevada, the threecorner milkvetch is found in Clark County along the Muddy River, the lower Virgin River, and Lake Mead (Overton Arm, Virgin Basin, Lower Basin) (Nevada Natural Heritage Program 2001). In Arizona, it is currently located in Mohave County in Sand Hollow Wash, Horsethief Canyon, and Beaver Dam Wash (Arizona Game and Fish Department 2005).

Populations within LCR MSCP Project Boundary
Threecorner milkvetch occurs within the LCR MSCP project boundary along Lake Mead at Middle Point and Ebony Cove (Virgin Basin), and Sandy Cove (Lower Basin) (Powell 1999, Powell 2003, Bangle 2005).

Life History

General Description
Threecorner milkvetch is a member of the legume (Asteraceae) family. It is a slender, ashy-pubescent herb, with stems 10-20 cm long. It has compound leaves 3-5 cm long, with approximately 9 elliptical, 4-15 mm long leaflets. Each raceme has 2-8 flowers. The corolla is white with pink veining (dries to violet), the banner petal is 5-8 mm long, and the keel petals are 4-5 mm long. The calyx is white and 2-4 mm long. Pods are 1 cm long, oblong, curved, and triangular in cross-section, with a groove on the lower side (AGFD 2005).

Phenology
The threecorner milkvetch flowers in April and May, with fruit setting in 4-6 weeks after flowering. Plants may be biennial or annual (Arizona Game and Fish Department 2005).

Habitat
Threecorner milkvetch is found on areas with stabilized sand, frequently with sparse gravel (AGFD 2005).

Threats

Major threats are off-road vehicle use away from Lake Mead and invasion of nonnative plants (Sahara mustard, Mediterranean grass, Russian thistle). Trampling by boaters along Lake Mead shoreline is considered a minor threat. Fluctuations in reservoir elevation are a potential threat for some populations (Bangle 2005).

LCR MSCP Conservation Measures

THMI1—Provide funding to support existing threecorner milkvetch conservation programs. The LCR MSCP will provide $10,000 per year until 2030 to the Clark County MSHCP Rare Plant Workgroup to support implementation of conservation measures for the threecorner milkvetch and sticky buckwheat that are beyond the permit requirements of the Clark County MSHCP.

LCR MSCP Research and Monitoring Needs

LCR MSCP conservation measures are restricted to providing funding to support existing threecorner milkvetch conservation programs. Research and monitoring will not be conducted through LCR MSCP implementation.

Other Research and Monitoring Opportunities

Several research needs have been identified by the National Park Service, including: 1) effects of dune movement on plant survival and germination, 2) effects of nonnative plants on dune movement, 3) environmental cues triggering germination and growth, 4) additional surveys during high-rainfall years, and 5) effects of competition with nonnative plants (Powell 1999, Bangle 2005).

Literature Cited

Arizona Game and Fish Department (AGFD). 2005. Heritage Data Management System: Plant Abstract. Available at http://www.azgfd.gov

Bangle, D. 2005. Report on *Astagalus geyeri* var. *triquetrus* (threecorner milkvetch) monitoring (2000-2005) within Lake Mead National Recreation Area. National Park Service. Unpublished report.

Nevada Natural Heritage Program. 2001. Nevada Rare Plant Atlas. Available at http://heritage.nv.gov/atlas/maps

Powell, E. 1998. Phenology of threecorner milkvetch, *Astragalus geyeri* var. *triquetrus*, at Sandy Cove, Lake Mead National Recreation Area. National Park Service. Unpublished report.

Powell, E. 1999. Report on 1997 and 1998 surveys for *Astragalus geyeri* var. *triquetrus* in Lake Mead National Recreation Area. National Park Service. Unpublished report.

Powell, E. 2003. Report on 2003 threecorner milkvetch (*Astragalus geyeri* var. *triquetrus*) monitoring on Sandy Cove, Lake Mead National Recreation Area. National Park Service. Unpublished report.

EVALUATION SPECIES

DESERT POCKET MOUSE
(*Chaetodipus penicillatus sobrinus*)

Introduction

The desert pocket mouse occurs throughout the deserts of the southwestern United States and northwestern Mexico. Two subspecies occur on the LCR, *Chaetodipus penicillatus sobrinus* and *Chaetodipus penicillatus penicllatus*. *Chaetodipus p. sobrinus* is the subspecies that is covered as an evaluation species under the LCR MSCP. The desert pocket mouse was previously classified under the scientific name *Perognathus penicillatus*, and is referred to by this scientific name prior to 1983.

Legal Status

C. p. sobrinus is an evaluation high priority species under the Clark County Multi-Species Habitat Conservation Plan. It is currently an evaluation species under the LCR MSCP habitat conservation plan.

Distribution

Chaetodipus penicillatus occurs in creosote bush and xeric riparian communities of the southwestern deserts of North America. The northern range encompasses areas from southeastern California, southern Nevada, and extreme southwest Utah. To the south, the species occurs to southeastern Baja California, Mexico, and the northern two-thirds of the state of Sonora, Mexico (Mantooth and Best 1995).

The *C. p. sobrinus* subspecies is restricted to an area that encompasses the Colorado, Virgin, and Muddy rivers in southeast Nevada and northwestern Arizona, as well as small populations in extreme southwestern Utah near Beaver Dam Wash (Hall 1946, Hoffmeister 1986). Currently, work is being done to delineate the exact range boundaries of the *sobrinus* subspecies. For instance, this subspecies does not seem to occur south of Hoover Dam. One possible population has been found south of Laughlin, Nevada, but its subspecies status has not yet been determined. (Zane Marshall pers. comm.). The other subspecies that occurs along the LCR, *C. p. penicillatus*, has a wider range and occurs from Topock on the LCR in the north, to Yuma, Arizona, in the south, and occurs eastward into Central Arizona, from south of the Mogollon Rim to San Carlos Reservoir (Hoffmeister 1986). Both subspecies are present on both sides of the LCR; the river has not served as a barrier to the distribution of this species. Pocket mice, including both LCR subspecies, occur in sandy areas, where vegetation is sparse (Hoffmeister 1986, Micone 2002). In the Las Vegas Valley, *C. p. sobrinus* was recorded for the first time in 1891 (Micone 2002), and not recorded again until 1997. Many of the extant populations of *C. p. sobrinus* are now isolated from one another, possibly due to human fragmentation of habitat (Micone 2002).

Life History

General Description

Chaetodipus p. sobrinus is the largest of the two subspecies occurring on the LCR, but is not strongly differentiated from *C. p. penincillatus* (Hoffmeister 1986). *Chaetodipus p. sobrinus* is differentiated, with difficulty, from *C. p. penincillatus* by greater body length, less divergent zygomatic arches posteriorly, mastoid breadth being relatively less, wider tips of nasals, more narrow rostrum at base, and a more pinkish, slightly lighter coloration (Hoffmeister 1986).

Chaetodipus p. sobrinus is the northernmost subspecies of pocket mouse. In studies conducted on this species in the Las Vegas Valley, activity was significantly correlated to minimum ambient temperature (Micone 2002). Observed, marked individuals became dormant during the winter, with a few individuals staying active. Individuals who stayed active may have been in subpar condition and unable to enter torpor due to a lack of energetic resources (Micone 2002). Maximum life span of individuals was 16 months, and the annual turnover of the population was estimated to be between 87 and 90% (Micone 2002). Recruitment occurred from June to early September. Females of the *C. p. sobrinus* subspecies tend to overlap their home ranges more than the males, which have demonstrated a higher level of territoriality (Micone 2002). *Chaetodipus p. sobrinus* showed a significant preference for coarse soils and habitats with a shrub component providing needed cover (Micone 2002).

Activity rates of *C. penicillatus* vary by season and temperature. In some cases, individuals enter into periods of torpor. The greatest period of activity occurs during late spring and early summer (Mantooth and Best 1995). In the northern part of the range, activity decreases greatly in winter months, with relatively fewer individuals captured from November to April (Bateman 1967, Van De Graaff 1975, Andersen and Nelson 1999). The species maybe be active year-round in the southern part of their range (Jameson and Peters 2004). In areas where winter activity decreases, some individuals enter into short periods of torpor during energetic crises (French 1993).

Breeding

The breeding season occurs from April to August, with peak reproductive activity occurring in June when 95% of males are capable of reproduction (Hoffmeister 1986, Mantooth and Best 1995). *Chaetodipus penicillatus* builds sphere-shaped nests of dry grass, dug to a depth of roughly 18 cm (Hoffmeister 1986). Gestation period is 26 days or more (Eisenberg and Isaac 1963, Wilken and Ostwald 1968). Litter size averages 3.38 (Van de Graaff 1975).

Habitat

Chaetodipus penicillatus occurs in desert areas with coarse vermiculite soils and clumped brush habitat (Price 1984, Mantooth and Best 1995). They tend to avoid more open desert areas, likely due to a lack of cover (Wondolleck 1978, Rosenzweig 1973, Price and Waser 1985). The general distribution of *C. penicillatus* corresponds to that of creosote (*Larrea*) and saltbush (*Atriplex*), and is strongly associated with the *Larrea-Atriplex* community (Hoffmeister and Lee 1967, Mantooth and Best 1995). They are the only species of heteromyid rodent commonly found in riparian woodland or tamarisk habitats (Stamp and Ohmart 1979). *Chaetodipus penicillatus* is fully independent of exogenous water (Grubbs 1974). While it prefers areas with shrubby canopy cover, *C. penicillatus* forages into open areas up to 4 m from cover (Rosenzweig 1973).

LCR MSCP Conservation Measures

DPMO1 – Conduct surveys to locate desert pocket mouse habitat.
Surveys are being conducted to locate desert pocket mouse habitat that could be affected by LCR MSCP habitat creation-related activities, to determine whether the habitat is occupied by the species. If the habitat is occupied, habitat creation-related activities will be designed to avoid the habitat. If the habitat cannot be avoided, to the extent practicable, the disturbed habitat area onsite will be restored following completion of the activities and the habitat protected and incorporated into the conservation area. If the habitat cannot be restored onsite, an amount of habitat will be created at least equal to the extent of disturbed habitat elsewhere in the conservation area. Restoring disturbed habitat will ensure that covered activities do not adversely affect the existing or potential future enhanced distribution, abundance, or population viability of the desert pocket mouse in the LCR MSCP planning area.

LCR MSCP Research and Monitoring Needs

Recently, a large amount of research has been conducted on the *C. p. sobrinus* subspecies, especially in the Las Vegas Valley. Recent graduate research has been conducted on *C. p. sobrinus* (Micone 2002, Marshall pers. comm.) and has gone a long way in delineating the habitat needs and areas of occurrence for this subspecies. Some questions still remain regarding the exact delineation of the range boundaries between the two subspecies that occur along the LCR, but current work is being conducted to answer this question. Little dedicated research needs to be conducted on *C. p. sobrinus* as part of the LCR MSCP program.

Most of the planned restoration efforts of the LCR MSCP in the near future are not going to take place near the range of *C. p. sobrinus*. However, general pre-monitoring of restoration sites for small mammals will be taking place before restoration implementation begins at each site.

Literature Cited

Andersen, D.C., and M. Nelson. 1999. Rodent use of anthropogenic and ‘natural’ desert riparian habitat, lower Colorado River, Arizona. Regulated Rivers: Research & Management 15:377-393.

Bateman, G.C. 1967. Home range studies of a desert nocturnal rodent fauna. Ph.D. Dissertation. University of Arizona, Tucson. 101 p.

Eisenberg, J.F., and D.E. Isaac. 1963. The reproduction of heteromyid rodents in captivity. Journal of Mammalogy 44:61-67.

French, A.R. 1993. Physiological ecology of the Heteromyidae: Economics of energy and water utilization. Pages 509-538 *In:* H. H. Genoways and J. H. Brown (editors), Biology of the Heteromyidae. Special Publication. The American Society of Mammalogists 10:1-719.

Grubbs, D.E. 1974. Desert rodents and desert agriculture: The ecological and physiological relationships of a desert rodent community to native and modified environments. Ph.D. Dissertation. University of California, Irvine, 135 p.

Hall, E.R. 1946. Mammals of Nevada. University of California Press, Berkeley.

Hoffmeister, D. 1986. Mammals of Arizona. University of Arizona Press, Tucson. 564 p.

Hoffmeister, D.F., and M.R. Lee. 1967. Revision of the pocket mice, *Perognathus penicillatus*. Journal of Mammalogy 48:361-380.

Jameson, E.W., and H.J. Peeters. 2004. California Mammals, Revised edition. University of California Press, Berkeley.

Longland, W.S. 1994. Effects of artificial bush canopies and illumination on seed patch selection by heteromyid rodents. American Midland Naturalist 132:82-90.

Mantooth, S.J., and T.L. Best. 2005. *Chaetodipus penicillatus*. Mammalian Species No. 767, p. 1-7. American Society of Mammalogists.

Micone, K. 2002. Demographics and ecology of an isolated population of the desert pocket mouse *Chaetodipus penicillatus sobrinus*, in the Las Vegas Valley, Nevada. Masters Thesis. University of Nevada, Las Vegas, 64 p.

Price, M.V. 1983. Laboratory studies of seed size and species selection by heteromyid rodents. Oecologia 60:259-263

Price, M.V. 1984. Microhabitat use in rodent communities: Predator avoidance or economics? Netherlands Journal of Zoology 34:63-80.

Price, M.V., and N.M. Waser. 1985. Microhabitat use by heteromyid rodents: Effects of artificial seed patches. Ecology 66:211-219.

Rosenzweig, M.L. 1973. Habitat selection experiments with a pair of coexisiting heteromyid rodent species. Ecology 54:111-117.

Stamp, N.E., and R.D. Ohmart. 1979. Rodents of desert shrub and riparian woodland habitats in Sonoran desert. Southwestern Naturalist 24:279-89.

Van De Graaff, K.M. 1975. Reproductive ecology of some Sonoran desert rodents. Ph.D. Dissertation, University of Northern Arizona, Flagstaff. 174 p.

Wilken, K.K., and R. Ostwald. 1968. Partial contact as a stimulus to laboratory mating in the desert pocket mouse, *Perognathus penicillatus*. Journal of Mammalogy 49:570-572.

Wondolleck, J.T. 1978. Forage-area separation and overlap in heteromyid rodents. Journal of Mammalogy 59:510-518.

CALIFORNIA LEAF-NOSED BAT (*Macrotus californicus*)

Legal Status

The California leaf-nosed bat is not Federally listed as threatened or endangered. The Bureau of Land Management (BLM) lists it as a sensitive species in California (BLM 2004). The State of California recognizes it as a mammalian species of special concern (Williams 1986). This bat was included in a draft list of Arizona Wildlife of Special Concern by the Arizona Game and Fish Department (AGFD 2001). The California leaf-nosed bat is a Nevada Species of Conservation Priority and is protected and considered sensitive (Nevada Department of Wildlife 2005). The Western Bat Working Group (1998) lists *M. californicus* as a species of “Red or High” priority, the highest priority they give. The International Union for Conservation of Nature (IUCN) red list of threatened species lists it as vulnerable, its third-highest rating (Chiroptera Specialists Group 1996).

Distribution

Historical Range

The historical range of *M. californicus* included records from San Diego County and Riverside County, California, eastward to Tombstone, Arizona, and south into Baja California and Sonora, Mexico, with the center of its distribution appearing to be the location of its first recorded description at Fort Yuma, California, opposite Yuma, Arizona (Grinnell 1918). Hatfield (1937) found leaf-nosed bats at a winter night roost east of Searchlight, Nevada, and Cockrum and Musgrove (1964) found a large roost in a mine 4.5 miles north of Davis Dam and 0.75 miles west of Lake Mojave. At least three mines that were known roost sites were inundated by water with the formation of Lake Mead and Lake Mojave (O’Farrell 1970). Later the species was found along the Colorado River at the extreme northwest corner of Arizona, as well as farther east to Glenbar, Graham County, Arizona. (Cockrum 1960 and Constantine1961).

Current Range

The current range includes southern Nevada, northwestern, central, and southwestern Arizona, and southwestern Chihuahua and Sinaloa, Mexico (Kays and Wilson 2002). A complete range map can be found at NatureServe.org (2006). Extensive surveys by Brown and Berry (1998, 2004) indicate that the California leaf-nosed bat’s range in California is now limited to only the eastern portion of the state, although a 2002-2003 bat survey in southwest San Diego County did record it at two different sites (Stokes et al. 2003). All records in Arizona were from below 4,000 feet (1,220 m) in elevation, with most below 2,500 feet (7,625 m) (AGFD 2001).

Populations within LCR MSCP Project Boundary

The Lower Colorado River Multi-Species Conservation Program (LCR MSCP 2004) project boundary includes all of the Colorado River from Separation Canyon in the lower end of the Grand Canyon to the Mexico border, including full pool elevations of the three main reservoirs

(Lake Mead, Lake Mojave, and Lake Havasu) along the LCR. The lower ends of the Virgin and Bill Williams rivers, which are LCR tributaries, are included in the LCR MSCP project area (LCR MSCP 2004).

The California leaf-nosed bat has known populations all along the LCR. In Nevada, it has been detected along the Muddy River in Moapa Valley, as well as the Las Vegas Wash, which drains runoff from Las Vegas into Lake Mead (Williams 2001 and O'Farrell 2006). In Lake Mead National Recreation Area (LMNRA), populations have been found roosting in three mines (Brown 2006). *Macrotus californicus* was captured on the Arizona side of LMNRA at a mine near Katherine Landing, which is now closed (Cockrum et al. 1996). Havasu National Wildlife Refuge (NWR) is the location of at least two mines that support leaf-nosed bats (Brown 2006 and Cockrum et al. 1996). Havasu NWR is located along approximately 30 miles of the Colorado River from Needles, California, to Lake Havasu City, Arizona. The Island Unit bridge at Cibola NWR was found to be an important night roosting spot for *M. californicus*, as well as other bat species. A mine near Hart Mine Wash was found to be one of the largest winter roosts for leaf-nosed bats along the LCR and is probably the day roost of the bats found at the Island unit bridge (Brown and Berry 2003). Cibola NWR is 15 miles south of Blythe California along the Colorado River. At least seven mines along the Bill Williams River contain colonies, ranging from 100 to 1000 *M. californicus* (Brown 1996). The Bill Williams River empties into Lake Havasu north of Parker, Arizona. During a survey done on the Arizona side of Imperial NWR, leaf-nosed bat roosts were found at 11 sites. Seven were known maternity sites, two were potential maternity sites, and two were bachelor colonies. Also during this survey, *M. californicus* was captured by mist netting at eight different desert wash locations (Castner et al. 1995a). A bat survey and inventory was conducted on the U.S. Army Yuma Proving Ground, adjacent to both Cibola NWR and Imperial NWR, by the Arizona Game and Fish Department (AGFD) in 1995. Eight sites had leaf-nosed bats present and 20 sites showed evidence of bat use, but no bats were present (Castner 1995b). There are 10 known maternity colonies found along the LCR with 7 of these considered major (more than 100 bats). There are also eight large winter roosts known of *M. californicus* along the LCR (Brown 2006).

Life History

General Description

California leaf-nosed bats originally were classified as their own species (Grinnell 1918). Later, they were classified as a subspecies (*Macrotus waterhousii californicus*) of a species of leaf-nosed bat found in Mexico, Guatemala, and the Caribbean (Anderson 1969). It regained species status when Davis and Baker (1974) found that *M. waterhousii* had a chromosomal diploid number of 2N = 46, while *M. californicus* had a chromosomal diploid number of 2N = 40. They also found cranial morphology to be different, and where range overlap occurred, there was no evidence of hybridization. A renal (kidney) morphology study found that *M. californicus* can utilize drier habitat than *M. waterhousii* because of their greater ability to concentrate urine and conserve water (Lu and Bleier 1981). The narrow range overlap between these two species appears to be limited by habitat preference.

The leaf-shaped nose is the most diagnostic character of this gray-furred, medium-sized bat. The ears are large (29-38 mm) and are joined near their base. The tail extends past the uropatagium 5-10 mm. The forearm measures 46-55 mm and weight varies from 12 to 22 g. *Choeronycteris mexicana* (Mexican long-tongued bat) and *Leptonycteris curasoae* (lesser long-nosed bat) have a similar nose projection but their ears are much smaller (less than 25 mm). Roosting leaf-nosed bats do not cluster in tight packs, as most other bat species (Hoffmeister 1986, Kays and Wilson 2002 and AGFD 2001). Total lifespan is not known; however, one *M. californicus* was recaptured after 15 years (Brown and Berry 1998). California leaf-nosed bats do not migrate or hibernate. They maintain a year-round presence by roosting in a cave or mine that maintains a high temperature (greater than 28ºC); many of these caves are geothermally heated.

Breeding

Females gather into maternity colonies in the spring and summer. These colonies usually range in size from 100 to 200 bats. Maternity colonies are either found in a different section of a mine/cave that is also used as a wintering site or one that is nearby. Males will roost separately but nearby to the maternity roost. Breeding takes place in the fall when males attract females with a courtship display that consists of wing-flapping and vocalizations. Males become territorial during these activities. After fertilization takes place, development of the embryo is delayed until the following spring. A single young is born between mid-May and early July (Hoffmeister 1986, Berry and Brown 1995, and AGFD 2001).

Diet

Echolocation and visual detection are used to locate prey, the latter used more in the winter months to save energy (Bell et al. 1986). Leaf-nosed bats feed by capturing prey during flight and by gleaning insects from vegetation. They primarily feed on large night flying beetles, grasshoppers, moths, and insect larvae, which they carry to a night roost to eat. These night roosts usually consist of shallow caves and short mining prospects that can be located by the accumulation of insect parts, such as wings, which are not eaten. Fruit-eating has also been reported (Huey 1925 and Hoffmeister 1986). Brown and Berry (2004) discovered a *M. californicus* at Havasu NWR feeding on a tree lizard (*Urosaurus ornatus*). Foraging normally takes place during the first 3 hours after sunset, as well as the last 2 hours before sunrise. An individual bat may forage for nearly 2 hours in a given night (AGFD 2001).

Habitat

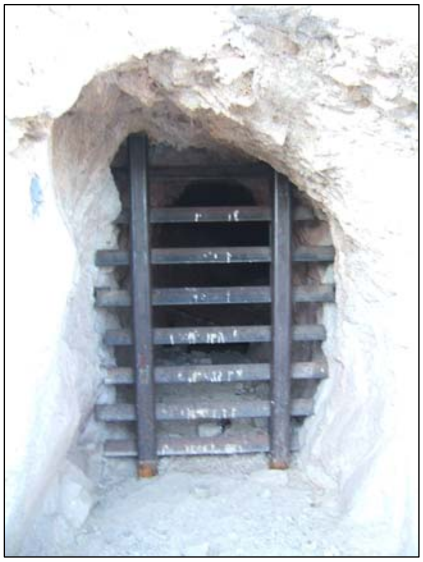
Foraging usually takes place in dry desert washes, 3-6 miles from the roost. In the winter, this distance decreases to one-half mile from the roost (Brown and Berry 1993, Brown 2005). Desert wash plant communities include: ironwood (*Olneya tesota*), paloverde (*Cercidium* spp.), mesquite (*Prosopis* spp.), catclaw (*Acacia greggii*), and smoketree (*Psorothamnus spinosa*) (Castner et al. 1995a). Huey (1925) found evidence of leaf-nosed bats utilizing riparian areas as well. Along the Muddy River, in Moapa Valley, Nevada, *M. californicus* was equally detected acoustically in four distinct habitat types: riparian marsh, mesquite bosques, riparian woodlands, and riparian shrublands. Woodlands consisted of Fremont cottonwood (*Populus fremontii*), velvet ash (*Fraximus velutina*), Goodding's willow (*Salix gooddingii*), and Washington fan palms *Washingtonia filifera*). Shrublands included stands of arrowweed (*Pluchea sericea*) and quailbush (*Atriplex lentiformis*) (Williams 2001). Foraging habitat is largely determined by insect abundance; therefore, it is understandable why leaf-nosed bats choose these sites as foraging habitat over the typical low desert scrub where their roosts are located. Desert scrub habitat of the Sonoran and Mojave deserts is dominated by creosote (*Larrea tridentata*) and bursage (*Ambrosia dumosa*). Roosting habitat is dependent on mines and caves that maintain high temperatures year-round. Because *M. californicus* forages during the entire year, foraging habitat in close proximity to roosting sites may be more important during the winter months (Brown and Berry 1993, Brown 2006). While night roosts consist of shallow caves in natural situations, manmade structures, including mining prospects, bridges, and buildings, are also readily used near foraging habitat (Huey 1925, Hatfield 1937, and Constantine 1961). The combination of day roosts, night roosts, and foraging habitat appears to be an important three-way association for a locale to support a population of California leaf-nosed bats.

Threats

The only specific example of predation on *M. californicus* is from evidence of a dentary bone in barn owl (*Tyto alba*) pellets in Sonora, Mexico (Bradshaw and Hayward 1960). Bats in general are preyed upon by a number of different animals, although most of these are not bat specialists and bats are usually a rare occurrence in their total diet. Known bat predators include domestic cats, dogs, birds of prey, snakes, raccoons, weasels, predatory song birds, frogs, large spiders, and even other bats (Fenton 2001). While humans are not predators of leaf-nosed bats, the negative image many have about bats may be a serious threat (Fenton 1997).

Disturbance and closure of roost mines are the greatest threats to the California leaf-nosed bat (Brown 2005). Disturbance may cause abandonment of roosts (AGFD 2001). The best way to keep a mine open for bats and safe for humans is to place a gate inside any and all entrances (Castner 1995a, AGFD 2001, Brown 2006). Bat gates allow bats and other wildlife to freely enter and exit a mine while restricting the access of humans (Figure 1). Because the bats are restricted to specific roost requirements (such as temperature), their limited distribution causes them to form a small number of large colonies rather than several small ones. The loss of one colony can have a significant effect on the total population along the LCR (Brown 2006).

Figure 1. Example of a gated mine at the Salt Creek Hills Mine near Baker, CA



LCR MSCP Conservation Measures

CLNB1—Conduct surveys to locate California leaf-nosed bat roost sites.
Conduct investigations to identify locations of California leaf-nosed bat roost sites within 5 miles of the LCR MSCP planning area in reaches 3-5.

CLNB2—Create covered species habitat near California leaf-nosed bat roost sites.
The LCR MSCP process for selecting sites to establish cottonwood-willow and honey mesquite as habitat for other covered species will, based on the information collected under conservation measure CLNB1, give priority to selecting sites that are within 5 miles of California leaf-nosed bat roosts in reaches 3-5, when consistent with achieving LCR MSCP goals for other covered species. As described in Section 5.4.3, created cottonwood-willow and honey mesquite land cover will be designed to establish stands that will support a substantially greater density and diversity of plant species that are likely to support a greater abundance of insect prey species than is produced in the affected land cover types (LCR MSCP 2004).

LCR MSCP Research and Monitoring Needs

The LCR MSCP conservation measures for the California leaf-nosed bat are designed to determine distribution along the LCR so that created habitat may be established within 5 miles of existing roosts, when this is consistent with achieving LCR MSCP goals for other covered species. Monitoring for foraging California leaf-nosed bats will occur through the multi-species bat monitoring at created habitat sites, as well as through system monitoring along the LCR.

California leaf-nosed bat use of habitat created by the LCR MSCP should be quantified. Distance between roost sites and created habitat will need to be compared to information for other foraging sites (e.g., desert washes) to determine whether proximity to roosts is a greater requirement (especially in winter) than the insect abundance of the created habitat. Habitat created near mines or caves that currently are not used by leaf-nosed bats but appear to be suitable roost sites should be monitored to determine whether those sites become roosts for bats. Because *M. californicus* uses low intensity echolocation, they are difficult to pick up on acoustic sampling equipment (Brown and Berry 2003, O’Farrell 2006). It may be necessary to mist net at restored habitat sites that are near known roosts to determine whether the bats are foraging in the habitat. These data may be used in the site selection process for determining habitat creation priorities when this is consistent with achieving LCR MSCP goals for other covered species.

Other Research and Monitoring Opportunities

LCR MSCP habitat creation projects will provide foraging habitat for California leaf-nosed bats. However, additional conservation can be achieved through the implementation of research and monitoring programs beyond the scope of the LCR MSCP conservation measures.

Research will determine the effectiveness of bat gates for protecting LCR populations by conducting an inventory of mines that have already been gated and prioritizing mines that have not been gated, based on the significance of the roost as well as the frequency of human disturbance in the area. Bat gate design protocols will need to be established to install gates that cannot be tampered with or destroyed (Brown and Berry 2003).

Las Vegas Wash bat monitoring currently consists of only stationary acoustic detectors. Because the detectors have detected leaf-nosed bats, it would be useful to increase monitoring to include mist netting, and determine foraging habitat of bat species using the wash. Habitat restoration is also occurring at Las Vegas Wash and many study designs could be tested on a small scale at the site. If successful, there would be a possibility of using this research on a larger scale along the entire LCR.

Renewed mining activities may occur in the future along the LCR. Determining a distance at which bat roosts may be impacted by these activities (including drilling and blasting) would help limit potential disturbance (Brown 2005). If mines are used by bats on a seasonal basis, avoidance measures may become necessary near known roost sites.

Literature Cited

Anderson, S. 1969. *Macrotus waterhousii*. Mammalian Species 1:1-4.

Arizona Game and Fish Department. 2001. *Macrotus californicus*. Unpublished abstract compiled and edited by the Heritage Data Management System, Arizona Game and Fish Department, Phoenix, Arizona. 6 p.

Bell, G.P., G.A. Bartholomew, and K.A. Nagy. 1986. The role of energetics, water, economy, foraging behavior, and geothermal refugia in the distribution of the bat, *Macrotus californicus*. Journal of Comparative Physiology B 156:441-450.

Berry, R.D., and P.E. Brown. 1995. [ABS]. Natural history and reproductive behavior of the California leaf-nosed bat (*Macrotus californicus*). Bat Research News 34:49-50.

Bradshaw, G. VR., and B. Hayward. 1960. Mammal skulls recovered from owl pellets in Sonora, Mexico. Journal of Mammalogy 41:282-283.

Brown, P.E. 1996. Bat survey of the Bill Williams River. Arizona Game and Fish Department, Heritage Fund Project I93073, administered by Lake Havasu, Bureau of Land Management.

Brown, P.E. 2005. *Macrotus californicus* California leaf-nosed bat. Western Bat Working Group. Accessed June 8, 2006. http://www.wbwg.org./species_accounts.htm#MACA.

Brown, P. 2006. Lower Colorado River bat monitoring protocol. Draft report submitted to Bureau of Reclamation. 30 p.

Brown, P.E., and R.D. Berry. 1998. The updated status and range of the California leaf-nosed bat (*Macrotus californicus*) in California. Report to California Department of Fish and Game.

Brown, P.E., and R.D. Berry. 2003. Baseline surveys and the development of monitoring protocol for lower Colorado River bat species. Report prepared for National Fish and Wildlife Foundation, Washington D.C., for the Lower Colorado River Multi-Species Conservation Program. Project #2000-0304-002. 76 p.

Brown, P.E., and R.D. Berry. 2004. Roost surveys and habitat requirements of rare southwestern bats: California leaf-nosed and Allen’s lappet-browed bats, with observations on Townsend’s big-eared and western mastiff bats. U.S. Geological Survey, Species at Risk Report 99HQAG0046. 58 p.

Brown, P.E., R.D. Berry, and C. Brown. 1993. [ABS]. Foraging behavior of the California leaf-nosed bat (*Macrotus californicus*) as determined by radio-telemetry. Bat Research News 34:104.

Bureau of Land Management. 2004. California – BLM animal sensitive species list, updated April 2004. Accessed June 9, 2006. http://www.blm.gov/ca/pdfs/pa_pdfs/biology_pdfs/SensitiveAnimals.pdf.

Castner, S.V., T.K. Snow, and D.C. Noel. 1995a. Bat inventory of the Imperial National Wildlife Refuge. Arizona Game and Fish Department. Technical Report 70. 30 p.

Castner, S.V., T.K. Snow, and D.C. Noel. 1995b. Bat inventory of the U.S. Army Yuma Proving Ground, Arizona: 1995. Arizona Game and Fish Department. Technical Report 90. 21 p.

Chiroptera Specialist Group. 1996. *Macrotus californicus*. *In*: IUCN 2006. 2006 IUCN Red List of Threatened Species. Accessed June 12, 2006. http://www.iucnredlist.org.

Cockrum, E.L. 1960. The recent mammals of Arizona: Their taxonomy and distribution. The University of Arizona Press, Tucson. p. 32-34.

Cockrum, E.L., and B. Musgrove. 1964. Cave myotis, *Myotis velifer*, from southern Nevada. Journal of Mammalogy 45:636-637.

Cockrum, E.L., B. Musgrove, and Y. Petryszyn. 1996. Bats of Mohave County, Arizona. Occasional Papers The Museum, Texas Tech University 157:1-71.

Constantine, D.G. 1961. Locality records and notes on western bats. Journal of Mammalogy 42:404-405.

Davis, B.L., and R.J. Baker. 1974. Morphometrics, evolution, and cytotaxonomy of mainland bats of the genus *Macrotus* (Chiroptera: Phyllostimatidae). Systematic Zoology 23:26-39.

Grinnell, H.W. 1918. A synopsis of the bats of California. University of California Publications in Zoology 17:252-257. University of California Press, Berkeley.

Fenton, M.B. 1997. Science and the conservation of bats. Journal of Mammalogy 78:1-14.

Fenton, M.B. 2001. Bats, revised edition. Checkmark Books, New York. p. 112-115.

Hatfield, D.M. 1937. Notes on the behavior of the California leaf-nosed bat. Journal of Mammalogy 18:96-97.

Hoffmeister, D.F. 1986. Mammals of Arizona. University of Arizona Press, Tucson. p. 59-62.

Huey, L.M. 1925. Food of the California leaf-nosed bat. Journal of Mammalogy 6:196-197.

Kays, R.W., and D.E. Wilson. 2002. Mammals of North America. Princeton University Press. Princeton, NJ. p. 140.

Lower Colorado River Multi-Species Conservation Program. 2004. Lower Colorado River Multi-Species Conservation Program, Volume II: Habitat Conservation Plan. p. 5-74

Lu, S.L., and W.J. Bleier. 1981. Renal morphology of *Macrotus* (Chiroptera, Phyllostomatidae). Journal of Mammalogy 62:181-182.

NatureServe. 2006. Comprehensive report species – *Macrotus californicus*. NatureServe Explorer (webpage). Accessed June 7, 2006. http://www.natureserve.org/explorer/servlet/NatureServe?searchName=Macrotus+californicus.

Nevada Department of Wildlife. 2005. Identification of species of conservation priority. State of Nevada Comprehensive Wildlife Conservation Strategy. p. 59.

O'Farrell Biological Consulting. 2006. Final report: Long-term acoustic monitoring of bat populations associated with an extensive riparian restoration program in Las Vegas Wash, Clark County, Nevada (2004-2005). Prepared for Southern Nevada Water Authority.

O'Farrell, M.J. 1970. Notes on the distribution of *Macrotus waterhouseii* in southern Nevada. The Great Basin Naturalist 30:53.

Stokes, D.C., C.S. Brehme, and R.N. Fisher. 2003. Bat inventory of the San Diego County multiple species conservation program area. U.S. Geological Survey Interim Report. 17 p.

Western Bat Working Group. 1998. Regional bat species priority matrix. Western Bat Working Group. Accessed April 6, 2006. http://www.wbwg.org/spp_matrix.html.

Williams, D.F. 1986. Mammalian species of special concern in California. California Department of Fish and Game. Accessed March 22, 2006. http://www.dfg.ca.gov/hcpb/info/mammal_ssc.shtml.

Williams, J.A. 2001. Community structure and habitat use by bats in the upper Moapa Valley, Clark County, Nevada. Unpublished M.A.S. Thesis. University of Nevada, Las Vegas. p. 55.

PALE TOWNSEND'S BIG-EARED BAT (*Corynorhinus townsendii*)

Legal Status

Two eastern subspecies of Townsend's big-eared bat (*C. t. ingens* and *C. t. virginianus*) have been listed by the U.S. Fish and Wildlife Service as endangered under the Endangered Species Act (USFWS 2006a, b). The Bureau of Land Management, in California, has placed *C. townsendii* on their animal sensitive species list (BLM 2004). The U.S. Forest Service places Townsend's big-eared bat on a sensitive animal species list for every forest in their Pacific Southwest Region (USFS 2001). State designations include mammalian species of special concern in California and a species of conservation priority by Nevada Department of Wildlife (NDOW) (Williams 1986, NDOW 2005). The Western Bat Working Group (WBWG 1998) lists *C. townsendii* as a species of "Red or High" priority, the highest priority the group gives. The International Union for Conservation of Nature (IUCN) red list of threatened species lists the species as vulnerable, its third-highest rating (Chiroptera Specialists Group 1996).

Distribution

Historical Range

Historically, three subspecies of Townsend's big-eared bat had a wide distribution across the West. Originally, these subspecies were separated by morphologic characters. *C. t. townsendii* was present in the western portions of California, Oregon, Washington, and British Columbia. The Pale Townsend's big-eared bat (*C. t. pallescens*) range included the eastern portions of those Pacific coast states and province, as well as all of Idaho, Nevada, Arizona, New Mexico, Utah, Wyoming, more than half of Montana, most of Colorado, western South Dakota, part of the Great Plains, and northwestern Mexico (not including the Baja Peninsula). The third subspecies (*C. t. australis*) distribution included extreme western Texas, and north central Mexico. Two additional subspecies have disjunct populations in central and eastern United States. The central subspecies (*C. t. ingens*) range includes southeastern Kansas, northeastern Oklahoma, northwestern Arkansas, and southwestern Missouri. The eastern subspecies (*C. t. virginianus*) range includes almost all of West Virginia, areas of Virginia that border West Virginia, and eastern Kentucky (Handley 1959).

Current Range

The current range of the species continues to include all areas where *C. townsendii* were historically found, although there have been population declines in many areas, including many historic roosting sites that no longer harbor Townsend's big-eared bat (Cockrum et al. 1996, Brown 2006). New evidence concerning the distribution of the western subspecies, *C. t. townsendii* and *C. t. pallescens,* has arisen. A recent DNA study found that *C. t. townsendii* is much more widely distributed than originally thought. The Pale Townsend's subspecies now appears to be restricted to central and eastern Colorado and most of New Mexico, except the southwestern corner. *C. t. townsendii* range now encompasses not only its original area, but all other areas that were formerly designated as *C. t. pallescens* range, except for central Colorado, eastern Colorado, and New Mexico (Piaggio and Perkins 2005). This may cause conservation measures in these areas to be modified.

Populations within LCR MSCP Project Boundary

The Lower Colorado River Multi-Species Conservation Program (LCR MSCP 2004) included the Pale Townsend's big-eared bat as an evaluation species on its covered species list. If this new subspecies evidence becomes widely accepted, it will mean that the lower Colorado River is in the range of *C. t. townsendii* rather than *C. t. pallescens*. The LCR MSCP project boundary includes all of the Colorado River from Separation Canyon, in the lower end of the Grand Canyon, to the Mexico border. This includes full pool elevations of the three main reservoirs (Lake Mead, Lake Mojave, and Lake Havasu) along the LCR. Because of full pool elevation, the lower ends of the Virgin and Bill Williams rivers, which are LCR tributaries, are included in the LCR MSCP project area (LCR MSCP 2004). There have been a number of historic roosting sites along the LCR. Usually mines included along the LCR are not actually in the project boundary; however, if the particular bat species' probable foraging areas are inside the boundary, they are considered LCR populations. Townsend's big-eared bat was first discovered along the LCR by Grinnell (1914) in a mine in the Riverside Mountains, west of the river. In 1916 and 1918, a mine north of Potholes, Imperial County, California, had a maternity colony (Howell 1920). A mine in Mohave County, Arizona, north of Davis Dam, had a maternity colony in the late 1950s and 1960s (Cockrum et al. 1996). Stager (1939) found big-eared bats to be common in another mine in the Riverside Mountains. None of these mines are now being used by *C. townsendii* (Brown 2006).

The only current site to have a known colony (less than 50 in 2003) along the LCR is a mine located in the Riverside Mountains (Brown 2004). There are two known roosting sites inside the Lake Mead National Recreation Area (NRA). One roost site is located west of Lake Mojave and the other is near Pearce Ferry at the upper end of Lake Mead. It is unknown if these populations forage along the LCR (P. Brown personal communication). Townsend's big-eared bats have been recorded acoustically from March through October at Las Vegas Wash, which empties runoff from the Las Vegas Valley into Lake Mead. Usually this bat is not picked up acoustically at distances farther than 10 miles from the recording device. Because this species was recorded at much higher rates than expected, there may be a population in the area, although no roosts are known at this time (O'Farrell 2006). Two maternity roosts have been found along the Bill Williams River, a major tributary that empties into Lake Havasu, north of Parker, Arizona (Brown 1996). Townsend's big-eared bats have also been observed in Moapa Valley, Nevada, near the Muddy River, which empties into the Overton Arm of Lake Mead (Williams 2001).

Life History

General Description

Nomenclature for C. townsendii has changed often since it was first described. From 1831 to 1897, the genera used for this species included *Synotus*, *Plecotus*, and *Corynorhinus*, the latter two being changed back and forth often (Miller 1897). Because of morphologic similarities, Handley (1959) revised the taxonomy and changed *Corynorhinus* to a subgenus and regrouped them with the Palearctic genus *Plecotus*. Cockrum (1960) continued this nomenclature. In the 1990s, three more detailed phylogenetic and morphologic studies were performed, which all concluded that *Corynorhinus* should be given back its full generic status for North American species (Frost and Timm 1992, Tumlison and Douglas 1992, and Bogdanowicz et al. 1998). The species name has also undergone many changes. Originally, all specimens of *Corynorhinus* were listed as one species (*C. macrotis*), with three different subspecies (*macrotis, pallescens*, and *townsendii*) designations (Miller 1897). *Macrotis* was later changed to *rafinesquii* for western individuals (Grinnell 1918). In 1955, new species designations were given, causing changes in species names for bats in the genus *Corynorhinus*. The original species name, *rafinesquii*, was changed to *townsendii*, while a southeastern big-eared bat (*C. macrotis*) was changed to *C. rafinesquii* (Handley 1955, 1959).

The Townsend's big-eared bat is a medium-sized bat with a wingspan of 30-34 cm, a forearm length of 3.9-4.7 cm, and a weight between 8-14 g. Dorsal hairs range from slate gray to pale with cinnamon brown to blackish brown tips that contrast slightly with the base. Ventral hairs are slate gray to brownish with brownish or buff tips. Ears are very large (30-39 mm) and are joined across the forehead. Hair on the toes does not project beyond the toenails. The most significant characteristics are two large glandular lumps on each side of the nose, which help distinguish it from the four other large-eared bat species that may be found along the LCR. These include the spotted bat (*Euderma maculatum*), whose pelage color is black with white spots, California leaf-nosed bat (*Macrotus californicus*), whose nose is shaped like a leaf, Allen's big-eared bat (*Idionycteris phyllotis*), which has small lappets projecting from the base of the ear, and the Pallid bat (*Antrozous pallidus*), whose ears are well separated. Roosting sites may be identified by their guano, which is usually found in circular patches in open areas (AGFD 2003, Kays and Wilson 2002).

Breeding

Breeding occurs in the hibernacula from October to February, although some individuals may mate prior to arriving. Males will perform a courtship ritual in which they emit twittering sounds while approaching a female and then rub the snout over the female's body. Males may copulate with hibernating females. Females may breed as early as 4 months of age. Males are not reproductively active until their second year. Females may mate with several males during the winter, and will store sperm until the spring when ovulation and fertilization occur. Maternity colonies form from March through April or later, depending on the elevation, and can range in size from 12 to 200 females in the western United States. Gestation lasts between 56 and 100 days so that between May and July a single young is born. The young are 25% of their mother's weight at birth, capable of flight at 2.5-3 weeks of age, and fully weaned at 6 weeks (Pearson et al. 1952, Pierson et al. 1999). The percentage of yearling females that returned to their natal site the following year was 38-54%; 75% of these females returned the following year, with 80% returning the year after (Pearson et al. 1952). From banding data, the two longest recorded life spans for the species are a 16-year, 5-month-old female, and a 21-year, 2-month-old male (Paradiso and Greenhall 1967, Perkins 1994).

Diet

Townsend's big-eared bat is considered a lepidopteran specialist because at least 90% of the diet is composed of moths. Other insects found to be preyed upon include: Coleoptera, Diptera, and

Hymenoptera (Sample and Whitmore 1993, Burford and Lacki 1995, 1998, Pierson et al. 1999). Generally, they take their prey in the air, although Howell (1920) noted evidence of foliage gleaning (Kunz and Martin 1982, Pierson et al. 1999). It is considered a slow flier and highly agile and maneuverable (Dalquest 1947, Hayward and Davis, Findley et al. 1972). This species leaves roosting sites to forage approximately 45 minutes after sunset (Clark et al. 1993). There have been two peaks of foraging activity observed, one right after leaving the roost, and a second that occurs close to sunrise the following morning (Cockrum and Cross 1964). Females in a maternity roost were recorded having three feeding periods throughout the night; they return to the roost after each feeding. As offspring matured, females decrease how often they returned to the roost; once the young mature, the females do not return until sunrise (Clark et al. 1993 and Clark et al. 2002).

Habitat

Foraging habitat varies widely between area and subspecies. Virginia big-eared bats (*C. t. virginianus*) were found to forage more in open fields, pastures, and cliffs, rather than in nearby forested areas (Sample and Whitmore 1993, Burford and Lacki 1995). Ozark big-eared bats (*C. t. ingens*) were found to use edge habitat or habitat in close proximity to vertical structures such as trees and cliffs more often than open field or woodland habitat. Open habitat was used more than woodland habitat during late lactation, but activity during early and mid-lactation did not differ statistically between the two (Clark et al. 1993). Townsend’s big-eared bats on Santa Cruz Island were found to avoid introduced vegetation near their roost, and travel 5 km to forage in a native oak (*Quercus* spp.) and ironwood (*Olneya tesota*) forest (Brown et al. 1994). A telemetry study at Point Reyes National Seashore found that *C. townsendii* concentrated foraging activity along the edges of riparian vegetation and generally were found in the vicinity of vegetation when traveling to foraging areas from the roost sites (Fellers and Pierson 2002). Foraging along edges is also thought to occur in northern Utah, where there is an interface between juniper woodlands and sagebrush-grass steppe. There appears to be an association between these foraging sites and the location of mines and caves that big-eared bats use as roosts (Sherwin et al. 2000). Along the LCR, most of the native riparian vegetation has been removed and replaced with agricultural fields. The Bill Williams River, which houses two large (>100 bats) maternity colonies, still contains large stands of native riparian vegetation.

Corynorhinus townsendii roosts exclusively in caves in the eastern United States and in caves, mines, old buildings and, in a few occurrences, large tree hollows in the western United States (Howell 1920, Dalquest 1947, Graham 1966, Burford and Lacki 1995, Pierson and Rainey 1998, Sherwin et al. 2000a and b, Fellers and Pierson 2002, Clark et al. 2002, Mazurek 2004). They can be found at a wide range of elevations from sea level to 2,400 m, with most records coming from around 915 m (Pierson et al. 1999 and AGFD 2003). Roost selection may be more complex than what is currently known (Sherwin et al. 2003). Site fidelity is considered high for maternity and winter roost sites, with 70-80% returning to the same site the following year (Pearson et al. 1952, Humphrey and Kunz 1976). Sometimes the use of an alternate roost occurs, possibly because of disturbance or an unknown factor (Pearson et al. 1952, Pierson and Rainey 1998, Pierson et al. 1999). Townsend’s big-eared bats generally do not associate with other bat species, especially in maternity roosts. A few individuals of other bat species may be present but not in direct contact with *C. townsendii*. Townsend’s big-eared bats form clusters on open surfaces of the roost site that are usually highly visible (Handley 1959).

Maternity roosts are known to house large groups of bats, ranging from as small as 17-40 bats in Kansas and Oklahoma to as large as 300-1,000 females farther east (Humphrey and Kunz 1976, Rippy and Harvey 1965, Pierson et al. 1999). Colonies in California average about 120 individuals, with the largest containing about 400 bats (Pierson and Rainey 1998). Roost temperature appears to be a factor in site selection for maternity colonies (Pearson et al. 1952, Lacki et al. 1994, Pierson and Rainey 1998). The colony tends to cluster to maintain body heat during pregnancy and lactation (Humphrey and Kunz 1976). In California, maternity roost sites vary from 19ºC in cooler areas to 30ºC in the warmer regions of southern California (Pierson et al. 1999). Having a constant temperature in a maternity roost may also be important. Mines in Mexico that house both *C. townsendii* and *C. mexicanus* were found to only have a difference in temperature of 6ºC or less in the spring, summer, and fall. These temperatures were taken during different times of the day. Interestingly, the temperature did drop dramatically during the winter months, which made the mine suitable for winter roosting. Most of the bats were found in mines that were at least 50 m in length (Lopez-Gonzalez, Torres-Morales 2004). Two Virginia big-eared bat maternity roost caves (one with two entrances) in Kentucky were measured with entrance openings of 0.53 m by 3.64 m, 2.42 m by 3.33 m and 2.18 m by 1.97 m. The room in one of the caves measured 6.06 m high and 9.39 m wide, while the other was not measured due to its large size and many internal passages, but had a ceiling of 1.7 m (Lacki et al. 1994). Small maternity colonies in Oklahoma and Kansas roosted in warmer portions of caves, with domes 7-12 m wide, or on large flat ceilings (Humphrey and Kunz 1976). Townsend's big-eared bats were found using basal hollows of redwood trees as maternity roosts, with 40-55 bats in the roost. These tree hollow roosts had openings of 4.7 m high and 1.5 m wide, with the interior being 2.92 m wide and 3.35 m deep (Mazurek 2004). Maternity sites in northern Utah were found to be more complex than bachelor roost sites, having larger entrances and more openings. Maternity roosts in caves were found to be larger and more spatially stable than those in mines. This is probably due to the fact that caves are an older, more dependable resource (Sherwin et al. 2000b). Site fidelity in the past has focused on the fidelity of one specific site. Research in Northern Nevada and Utah points to much variation in movement at sites on a short-term scale (within a season), but on a longer scale (from year to year), patterns of movement have shown that, if bats have moved from one site, they may reliably be found at another nearby site. This was found most often in bachelor roosts, but also found to be common for maternity and winter colonies. Compared to cave roosts, movement in mine roosts was found to be greater, especially for bachelor roosts (Sherwin et al. 2000a). Criteria established for *C. townsendii* maternity roosts in California include (Pierson and Rainey 1998):

- Roost entrance minimum size of 15 cm high and 31 cm wide
- Roost height size minimum of 1.0 m with an average of 2.5-5.0 m
- Roost area minimum large enough for flying forays
- Light quality of semi-dark to dark
- Temperature of 18º-30ºC
- Humidity of 19-93% (relative humidity)
- Distance to water of within 100 m for coastal populations and 8 km for others

Unlike maternity colonies, bachelor (and non-reproductive female) roosting sites usually contain one to several individuals, although one site in Kentucky had more than 1,000 bats together in a bachelor roost (Pierson and Rainey 1998, Pierson et al. 1999, Sherwin et al. 2000b, and Lacki et al. 1994). Humphrey and Kunz (1976) found a maximum of six males in a roost together, with an average of two bats, in a total of 25 caves. Along the LCR, males may be territorial and roost alone unless the site is very large (P. Brown personal communication). Bachelor roost selection is not as complex as it is for maternity colonies (Humphrey and Kunz 1976, Lacki et al. 1994, Sherwin et al. 2000b). Similar to maternity sites, Sherwin et al. (2000a, b) found bachelor sites more temporally stable in caves than in mines, with an 89% chance of finding a bat on a subsequent night in caves compared to only a 38% chance of finding a bat at a mine roost. In Kentucky, the large bachelor colonies begin to break up around the end of summer with the onset of breeding that generally occurs throughout the fall before hibernation begins (Lacki et al. 1994).

Night or feeding roosts are also used by big-eared bats. Night roosts are usually found much closer to feeding areas because these roosts main use is for a place to feed on large prey items that cannot be eaten in mid-flight. Bats generally don't form large groups in night roosts (Pierson et al. 1999). Feeding roosts of most species can be identified by a culmination of insect body parts (mainly moth wings for *C. townsendii*) on the floor of the roost. These insect parts are used to collect information on the prey eaten by bats (Lacki et al. 1993). In Kentucky, researchers found that 45 species of moths were consumed by *C. townsendii virginianus* in the area. This also enabled them to learn more about foraging habitat by what type of habitat the moths generally use (Burford and Lacki 1998). Characteristics of feeding roosts have been found to be highly variable. In Kentucky, big-eared bats used cliff shelters with large entrances and deep passages as night roosts (Lacki et al. 1993).

Winter roosting sites, or hibernacula, in the westen United States generally consist of aggregations of a few to several dozen males and females, although sites with a single bat have been found (Humphrey and Kunz 1976, Kunz and Martin 1982, and Pierson et al. 1999). Larger groups, up to 1,000 individuals, are more common in the eastern United States probably because suitable wintering sites are limited (Rippy and Harvey 1965, Pierson et al. 1999). In the West, aggregations numbering greater than 400 have been found in colder areas (Pierson and Rainey 1998). *Corynorhinus townsendii* begin to arrive at the hibernacula in October and reach a maximum number of individuals in January. In early winter, they may roost near the entrance, but if temperatures drop below freezing, they will move into deeper, more stable parts of the cave or mine (Kunz and Martin 1982). When hibernating, they are known to cluster and curl their ears when the temperature drops. Females have been found to inhabit colder winter sites than males (Pearson et al. 1952). Townsend's big-eared bats are known to have periods of activity in the winter, although feeding has yet to be confirmed (Pearson et al. 1952, Bosworth 1994, Pierson and Rainey 1998, Clark et al. 2002). Winter activity in Idaho decreases in January and February and begins to increase again until the end of hibernation (Bosworth 1994). In the West, *C. townsendii* selects roosts with cold, stable temperatures and moderate airflow (Humphrey and Kunz 1976, Kunz & Martin 1982). Temperatures have been found to range from −2.0-13.0ºC, with temperatures below 10ºC preferred (Pearson et al. 1952, Twente 1955, Humphrey and Kunz 1976, Pierson and Rainey 1998). Unlike maternity sites, at least 11 other bat species have been found sharing *C. townsendii* hibernacula (Dalquest 1947, Pearson et al. 1952, Twente 1955, Handley 1959, Rippy and Harvey 1965, Kunz and Martin 1982, Pierson et al. 1999).

Threats

Threats can be separated into natural and human caused. The natural behavior to gather in large aggregations may be a threat to a population if that roost is disturbed. The low (38-54%) return rate of yearling females to the maternity roost is a sign of low reproductive potential. A dramatic decrease in reproductive females may cause a population to take an extended period of time to recover. There is a possibility that gene flow among populations may be low because of their sedentary behavior, which may be exacerbated when maternity colonies are small (Pierson et al. 1999).

Predation is a threat to most bats, including Townsend’s big-eared bats. Specific predators of *C. townsendii* include black rat snake (*Elaphe obsolete*), spotted skunk (*Spilogale putorius*), house cats (*Felis catus*), ringtails (*Bassariscus astutus*), and black rats (*Rattus rattus*) (Pearson et al. 1952, Pierson et al. 1999, and Fellers 2000). Bats, in general, are preyed upon by a number of different animals, although most of these are not bat specialists and bats are usually a rare occurrence in their total diet. Known bat predators include: domestic cats, dogs, birds of prey, snakes, raccoons, weasels, predatory song birds, frogs, large spiders, and even other bats (Fenton 2001). While humans are not predators of bats, the negative image many have about bats may be a serious threat (Fenton 1997).

Human-caused disturbances occur in a variety of different ways. The loss of roosting habitat for this sedentary species may be one of the most serious threats to not only *C. townsendii*, but other species as well (Pierson et al. 1999). Townsend’s big-eared bats lose roosting habitat by either the destruction of the roost or by abandonment after a disturbance. In some areas where *C. townsendii* are found, mines are the only sites being used for roosting habitat. In the past, mines were closed with no regard to the benefit they give to wildlife (Pierson and Rainey 1998, Pierson et al. 1999). Today it is more common for mines to be evaluated for wildlife use. Bat gates can be placed at mine openings to keep humans out and still allow bats and other wildlife to use the mine (figure 1). Townsend’s big-eared bat populations have been found to increase rapidly after the installation of a bat gate (Sherwin et al. 2002). Renewed mining of an abandoned mine will also cause a mine to become unacceptable, especially when the renewed operation uses open pit mining practices (Pierson et al. 1999). Caves have also been altered by being incorporated into mine operations (M. Wilkins, personal communication in Pierson et al. 1999).

Figure 1. Example of a gated mine at the Salt Creek Hills Mine near Baker, CA

Disturbance to maternity roosting sites has been found to be a serious danger to Townsend's big-eared bat populations (Pearson et al. 1952, Graham 1966, Humphrey an Kunz 1976, Kunz and Martin 1982, and Pierson and Rainey 1998). Disturbances at hibernacula may also be a danger because it causes an increase in activity, which may cause bats to expend too much energy, causing them to starve to death (Pearson et al. 1952, Twente 1955, Humphrey and Kunz 1976, Pierson et al. 1999). Cave and mine explorers, and well-intentioned scientists can have adverse affects on bat populations (Pierson et al. 1999).

Pesticide spraying can greatly decrease the insect prey base. Non-target spraying that affects large areas are the most common spray techniques in the West (Pierson et al. 1999). In the East, sprays that target gypsy moths also tend to lower numbers of other moth species, in turn decreasing the prey base for moth specialists such as *C. townsendii* (Sample and Whitmore 1993). Conversion of native habitat to agriculture and grazing lands also threatens foraging habitat for bats. Proximity of good foraging habitat may be a determining factor in roost selection. Brown (2006) has observed that *C. townsendii* in the Panamint Mountains would roost in suitable mines if they were within 3.2 km of a canyon with water. It is thought that a combination of land conversion and pesticide use on converted land contributes to the decrease in insect prey. In some areas, timber harvesting may impact bat populations. For example, the latest evidence of *C. townsendii* using hollows of redwood trees in California may be important to forest management in those areas (Pierson et al. 1999, Mazurek 2004).

Threats specific to the LCR include both disturbance of roosts and foraging habitat. Mines and caves along the LCR are known to be highly used for recreational purposes. The loss of native vegetation and the extensive spraying of agricultural fields are probably to blame for population declines along the LCR (Pierson and Rainey 1998, Brown 2006).

LCR MSCP Conservation Measures

PTBB1—Conduct surveys to locate pale Townsend's big-eared bat roost sites.
Conduct investigations to identify locations of pale Townsend's big-eared bat roost sites within 10 miles of the LCR MSCP planning area in reaches 3-5.

PTBB2—Create covered species habitat near pale Townsend's big-eared bat roost sites.
The LCR MSCP process for selecting sites to establish cottonwood-willow and honey mesquite as habitat for other covered species will give priority to selecting sites that are within 10 miles of pale Townsend's big-eared bat roosts in reaches 3-5, when consistent with achieving LCR MSCP goals for other covered species, based on information collected under conservation measure PTBB1. As described in Section 5.4.3, created cottonwood-willow and honey mesquite land cover will be designed to establish stands that will support substantially greater density and diversity of plant species that are likely to support a greater abundance of insect prey species than currently produced in the affected land cover types (LCR MSCP 2004).

LCR MSCP Research and Monitoring Needs

LCR MSCP conservation measures for the pale Townsend's big-eared bat are designed to determine distribution along the LCR so that created habitat may be established within 10 miles of existing roosts, when this is consistent with achieving LCR MSCP goals for other covered species. Monitoring for foraging pale Townsend's big-eared bats will occur through the multi-species bat monitoring at created habitat sites, as well as through system monitoring along the LCR.

Foraging habitat characteristics are not well understood along the LCR. Habitat characteristics, including type and structure of vegetation, as well as distance between maternity roosts and foraging sites, must be determined. Insect diversity and abundance within created habitat and known foraging habitat along the LCR should be compared to determine whether created habitat is providing the prey base necessary for foraging Townsend's big-eared bats. Pesticide use adjacent to sites may also be important to record because pesticides may lower prey abundance and may even have a negative health effect on the bats themselves. Light traps can be used in specific habitat types to monitor nocturnal insect diversity and abundance. It should be noted that it may be necessary to use a variety of different colored lights in the traps to gain the highest diversity of insects found at a site (Burles and Ring 2005).

Very little is known about roosting requirements along the LCR. The location of the hibernacula for the Riverside Mountains population is not known. Currently, the mine used as a maternity roost has been approved for gating and is awaiting installation (P. Brown personal communication). Discovering the location of the hibernacula will allow for both maternity and wintering roosts to be protected.

Other Research and Monitoring Opportunities

LCR MSCP habitat creation projects will provide foraging habitat for pale Townsend's big-eared bats. However, additional conservation can be achieved through the implementation of research and monitoring programs beyond the scope of the LCR MSCP conservation measures.

Las Vegas Wash bat monitoring currently utilizes stationary acoustic detectors. Because Southern Nevada Water Authority detected *C. townsendii*, it would be useful to increase monitoring by mist netting, which could be used to determine foraging behavior of bat species using the wash. Roosting sites for these Townsend's big-eared bats are unknown. The location of both maternity and wintering roosts needs to be determined to assure protection of those sites. Once this is determined, foraging habitat should also be studied and compared with the populations along the Bill Williams River and in the Riverside Mountains.

Foraging habitat at two sites in Lake Mead NRA needs to be determined. Wintering roosts also need to be discovered. Roosts need to be evaluated to determine if they should be gated. These roost sites are located in reaches 1 and 2 of the LCR MSCP planning area. Habitat creation activities are not included in reaches 1 and 2 for this species because of the lack of suitable areas to create habitat. Conservation opportunities in these areas may include protecting roost sites and conserving current foraging areas. Population genetics within *C. townsendii* needs to be better understood. Gene flow may be highly restricted for *C. townsendii* in all areas (Pierson et al. 1999). Because most roosts near the LCR are isolated from each other, it is important to identify possible genetic bottlenecks.

Literature Cited

Arizona Game and Fish Department. 2003. *Corynorhinus townsendii pallescens*. Unpublished abstract compiled and edited by the Heritage Data Management System, Arizona Game and Fish Department, Phoenix, Arizona. 6 p.

Bogdanowicz, W., S. Kasper, and R.D. Owen. 1998. Phyologeny of plecotine bats: Reevaluation of morphological and chromosomal data. Journal of Mammalogy 79:78-90.

Bosworth, W.R. 1994. Characteristics of winter activity in *Plecotus townsendii* in southeastern Idaho. Thesis. Idaho State University, Pocatello, Idaho.

Brown, P.E. 1996. Bat survey of the Bill Williams River. Arizona Game and Fish Department, Heritage Fund Project I93073, administered by Lake Havasu Bureau of Land Management.

Brown, P. 2006. Lower Colorado River bat monitoring protocol. Draft report submitted to Bureau of Reclamation. 30 p.

Brown, P.E., and R.D. Berry. 2004. Roost surveys and habitat requirements of rare southwestern bats: California leaf-nosed and Allen’s lappet-browed bats, with observations on Townsend’s big-eared and western mastiff bats. U.S. Geological Survey, Species at Risk Report 99HQAG0046. 58 p.

Brown, P.E., R.D. Berry, and C. Brown. 1994. Foraging behavior of Townsend’s big-eared bats (*Plecotus townsendii*) on Santa Cruz Island. Pages 367-369 *In*: W. L. Halvorson and G. J. Maender (editors), Fourth California Islands Symposium, Santa Barbara Museum of Natural History, Santa Barbara, California.

Bureau of Land Management. 2004. California – BLM animal sensitive species list, updated April 2004. Accessed June 9, 2006. http://www.blm.gov/ca/pdfs/pa_pdfs/biology_pdfs/SensitiveAnimals.pdf.

Burford, L.S., and M.J. Lacki. 1995. Habitat use by *Corynorhinus townsendii virginianus* in the Daniel Boone National Forest. American Midland Naturalist 134:340-345.

Burford, L.S., and M.J. Lacki. 1998. Moths consumed by *Corynorhinus townsendii virginianus* in eastern Kentucky. American Midland Naturalist 139:141-146.

Burles, D.W., and R.A. Ring. 2005. Which colour of light should you use to monitor insect abundance? Western Bat Working Group Abstracts From the 2005 Biennial Meeting. Accessed January 6, 2006. http://www.wbwg.org/abstract.htm.

Chiroptera Specialist Group. 1996. *Plecotus townsendii*. *In*: IUCN 2006. 2006 IUCN Red List of Threatened Species. Accessed July 21, 2006. http://www.iucnredlist.org.

Clark, B.K., B.S. Clark, and D.M. Leslie. 1997. Seasonal variation in use of caves by the Endangered Ozark big-eared bat (*Corynorhinus townsendii ingens*) in Oklahoma. American Midland Naturalist 137:388-392.

Clark, B.S., B.K. Clark, and D.M. Leslie. 2002. Seasonal variation in activity patterns of the Endangered Ozark big-eared bat (*Corynorhinus townsendii ingens*). Journal of Mammalogy 83:590-598.

Clark, B.S., D.M. Leslie, and T.S. Carter. 1993. Foraging activity of adult female Ozark big-eared bats (*Plecotus townsendii ingens*) in summer. Journal of Mammalogy 74:422-427.

Cockrum, E.L. 1960. The recent mammals of Arizona: Their taxonomy and distribution. The University of Arizona Press, Tucson. p. 56-59.

Cockrum, E.L., and S.P. Cross. 1964. Time of bat activity over water holes. Journal of Mammalogy 45:635-636.

Cockrum, E.L., B. Musgrove, and Y. Petryszyn. 1996. Bats of Mohave County, Arizona. Occasional Papers The Museum, Texas Tech University 157:1-71.

Dalquest, W.W. 1947. Notes on the natural history of the bat *Corynorhinus rafiesquii* in California. Journal of Mammalogy 28:17-30.

Fellers, G.M. 2000. Predation on *Corynorhinus townsendii* by *Rattus rattus*. The Southwestern Naturalist 45:524-527.

Fellers, G.M., and E.D. Pierson. 2002. Habitat use and foraging behavior of Townsend’s big-eared bat (*Corynorhinus townsendii*) in coastal California. Journal of Mammalogy 83:167-177.

Fenton, M.B. 1997. Science and the conservation of bats. Journal of Mammalogy 78:1-14.

Fenton, M.B. 2001. Bats, revised edition. Checkmark Books, New York, New York. p. 112-115.

Findley, J.S., E.H. Studier, and D.E. Wilson. 1972. Morphologic properties of bat wings. Journal of Mammalogy 53:429-444.

Frost, D.R., and R.M. Timm 1992. Phylogeny of plecotine bats (Chiroptera: “Vespertilionidae”): Summary of the evidence and proposal of a logically consistent taxonomy. American Museum Novitates 3034:1-16.

Graham, R.E. 1966. Observations on the roosting habits of the big-eared bat, *Plecotus townsendii*, in California limestone caves. Cave Notes 8:17-22.

Grinnell, J. 1914. An account of the mammals and birds of the lower Colorado Valley. University of California Publications in Zoology 12:51-294.

Grinnell, H.W. 1918. A synopsis of the bats of California. University of California Publications in Zoology 17:340-347. University of California Press, Berkeley.

Handley, C.O. 1955. New bats of the genus *Corynorhinus*. Journal of the Washington Academy of Sciences 45:147-149.

Handley, C.O. 1959. A revision of American bats of the genera *Euderma* and *Plecotus*. Proceedings of the U.S. National Museum 110:95-246.

Hayward, B., and R. Davis. 1964. Flight speeds in western bats. Journal of Mammalogy 45:236-242.

Howell, A.B. 1920. Some Californian experiences with bat roosts. Journal of Mammalogy 1:169-177.

Humphrey, S.R., and T.H. Kunz. 1976. Ecology of a Pleistocene relict, the western big-eared bat (*Plecotus townsendii*), in the southern Great Plains. Journal of Mammalogy 57:470-494.

Kays, R.W., and D.E. Wilson. 2002. Mammals of North America. Princeton University Press. Princeton, NJ. p. 146.

Kunz, T.H., and R.A. Martin 1982. *Plecotus townsendii*. Mammalian Species 175:1-6.

Lacki, M.J., M.D. Adam, and L.G. Shoemaker. 1993. Characteristics of feeding roosts of Virginia big-eared bats in Daniel Boone National Forest. Journal of Wildlife Management 57:539-543.

Lacki, M.J., M.D. Adam, and L.G. Shoemaker. 1994. Observations on seasonal cycle, population patterns and roost selection in summer colonies of *Plecotus townsendii virginianus* in Kentucky. American Midland Naturalist 131:34-42.

Lopez-Gonzalez, C., and L. Torres-Morales. 2004. Use of abandoned mines by long-eared bats, genus *Corynorhinus* (Chiroptera: Vespertilionidae) in Durango, Mexico. Journal of Mammalogy 85:989-994.

Lower Colorado River Multi-Species Conservation Program. 2004. Lower Colorado River Multi-Species Conservation Program, Volume II: Habitat Conservation Plan. p. 5-74 through 5-75.

Mazurek, M.J. 2004. A maternity roost of Townsend’s big-eared bats (*Corynorhinus townsendii*) in coast redwood basal hollows in northwestern California. Northwestern Naturalist 85:60-62.

Miller, G.S. 1897. Revision of the North American bats of the family Vespertilionidae. North American Fauna 13. 135 pages.

Nevada Department of Wildlife. 2005. Identification of species of conservation priority. State of Nevada Comprehensive Wildlife Conservation Strategy. p. 59.

O'Farrell Biological Consulting. 2006. Final report: Long-term acoustic monitoring of bat populations associated with an extensive riparian restoration program in Las Vegas Wash, Clark County, Nevada (2004-2005). Prepared for Southern Nevada Water Authority.

Paradiso, J.L., and A.M. Greenhall 1967. Longevity records for American bats. American Midland Naturalist 78:251-252.

Pearson, O.P., M.R. Koford, and A.K. Pearson. 1952. Reproduction of the lump-nosed bat (*Corynorhinus rafinesquei*) in California. Journal of Mammalogy 33:273-320.

Perkins, J.M. 1994. Longevity records for two vespertilionids. Bat Research News 35:79-80.

Piaggio, A.J., and S.L. Perkins 2005. Molecular phylogeny of North American long-eared bats (Vespertilionidae: *Corynorhinus*); inter- and intraspecific relationships inferred from mitochondrial and nuclear DNA sequences. Molecular Phylogenetics and Evolution 37:762-775.

Pierson, E.D., and W.E. Rainey. 1998. Distribution, status, and management of Townsend's big-eared bat (*Corynorhinus townsendii*) in California. California Department of Fish and Game, Wildlife Management Division, Bird and Mammal Conservation Program Report 96-7, Sacramento, California.

Pierson, E.D., M.C. Wackenhut, J.S. Altenbach, P. Bradley, P. Call, D.L. Genter, C.E. Harris, B.L. Keller, B. Lengus, L. Lewis, B. Luce, K.W. Navo, J.M. Perkins, S. Smith, and L. Welch. 1999. Species conservation assessment and strategy for Townsend's big-eared bat (*Corynorhinus townsendii townsendii* and *Corynorhinus townsendii pallescens*). Idaho Conservation Effort, Idaho Department of Fish and Game, Boise, Idaho.

Rippy, C.L., and M.J. Harvey. 1965. Notes on *Plecotus townsendii virginianus* in Kentucky. Journal of Mammalogy 46:499.

Sample, B.E., and R.C. Whitmore. 1993. Food habits of the endangered Virginia big-eared bat in West Virginia. Journal of Mammalogy 74:428-435.

Sherwin, R.E., J.S. Altenbach, and S. Haymond. 2002. The responses of bats to gates. Office of Surface Mining, Mid-Continent Region. Accessed July 21, 2006. http://www.mcrcc.osmre.gov/PDF/Forums/Bat%20Gate%20Design/6-1.pdf.

Sherwin, R.E., W.L. Gannon, and J.S. Altenach. 2003. Managing complex systems simply: Understanding inherent variation in the use of roosts by Townsend's big-eared bat. Wildlife Society Bulletin 31:62-72.

Sherwin, R.E., W.L. Gannon, J.S. Altenbach, and D. Strickland. 2000a. Roost fidelity of Townsend's big-eared bat in Utah and Nevada. Transactions of the Western Section of the Wildlife Society 36:15-20.

Sherwin, R.E., D. Strickland, and D.S. Rogers. 2000b. Roosting affinities of Townsend's big-eared bat (*Corynorhinus townsendii*) in Northern Utah. Journal of Mammalogy 81:939-947.

Stager, K.E. 1939. Status of *Myotis velifer* in California, with notes on its life history. Journal of Mammalogy 20:225-228.

Tumlison, R., and M.E. Douglas. 1992. Parsimony analysis and the phylogeny of the plecotine bats (Chiroptera: Vespertilionidae). Journal of Mammalogy 73:276-285.

Twente, J.W. 1955. Some aspects of habitat selection and other behavior of cavern-dwelling bats. Ecology 36:706-732.

U. S. Fish and Wildlife Service. 2006a. Bat, Ozark big-eared. Accessed July 21, 2006. http://ecos.fws.gov/species_profile/servlet/gov.doi.species_profile.servlets.SpeciesProfile?spcode=A075.

U. S. Fish and Wildlife Service. 2006a. Bat, Virginia big-eared. Accessed July 21, 2006. http://ecos.fws.gov/species_profile/servlet/gov.doi.species_profile.servlets.SpeciesProfile?spcode=A080.

U.S. Forest Service. 2001. USDA Forest Service, Pacific Southwest Region, Sensitive Animal Species by Forest. Accessed July 21, 2006. http://www.fs.fed.us/r5/projects/sensitive-species/sensitive-animals.pdf.

Western Bat Working Group. 1998. Regional bat species priority matrix. Western Bat Working Group (webpage). Accessed April 6, 2006. http://www.wbwg.org/spp_matrix.html.

Williams, D. F. 1986. Mammalian species of special concern in California. California Department of Fish and Game (webpage). Accessed March 22, 2006. http://www.dfg.ca.gov/hcpb/info/mammal_ssc.shtml.

Williams, J. A. 2001. Community structure and habitat use by bats in the upper Moapa Valley, Clark County, Nevada. Unpublished M.A.S. Thesis. University of Nevada, Las Vegas. p. 55.

COLORADO RIVER TOAD
(*Bufo alvarius*)

Legal Status

Bufo alvarius is not Federally listed. *Bufo alvarius* is listed as threatened in New Mexico (Degenhardt et al. 1996). Natureserve ranks *Bufo alvarius* as secure on a national and global level. Natureserve ranks *Bufo alvarius* as secure in Arizona, imperiled in New Mexico, and possibly extirpated in California (NatureServe 2006).

Distribution

Historical Range

The westernmost record of *Bufo alvarius* for Sonora, Mexico, is 9.4 miles east of Huasabas (Wright 1966). California records were restricted to bottomlands and irrigated areas of the Colorado River Delta region in Imperial County (Grinnell and Camp 1917, Storer 1925, Slevin 1928). Other historical records include Tiburin Island (Malkin 1962); 4.7 miles north of El Mayor in Baja, California (Brattstrom 1961); southwestern New Mexico (Cole 1962); northwest of the junction of Arizona Highway 71 on US Highway 93, Mohave and Yavapai counties (Fouquette 1970); Fort Mohave, California (Cooper 1869; Mearns 1907); Phoenix, Arizona (Musgrave and Cochran 1930); 27.5 miles east of Douglas in Guadalupe Canyon at Boundary Survey Monument No. 73 on the border of Cochise County, Arizona, and Sonora, Mexico (Mearns 1907); 27.5 miles east of Douglas at San Bernardino Springs near Boundary Survey Monument No. 77 in Cochise County, Arizona (Mearns 1907); Camp Grant, Graham County, Arizona (Slevin 1928); New Mexico, Hildago County (Cole 1962) and southwest of Tucson, Arizona (Arnold 1943, Kauffeld 1943).

The historical range of *Bufo alvarius* in California extended along the floodplain of the lower Colorado River (LCR) and in the southern Imperial Valley. Historically, *Bufo alvarius* was documented on the LCR from Fort Yuma to the Blythe-Ehrenberg Region (Fouquette 1968, Vitt and Ohmart 1978). The range likely extended along the LCR to extreme southern Nevada, near Fort Mohave (Mearns 1907, Storer 1925, Stebbins 1951). The lower Colorado River Multi-Species Conservation Program (LCR MSCP) planning area is in on the western edge of the historical range of *Bufo alvarius*.

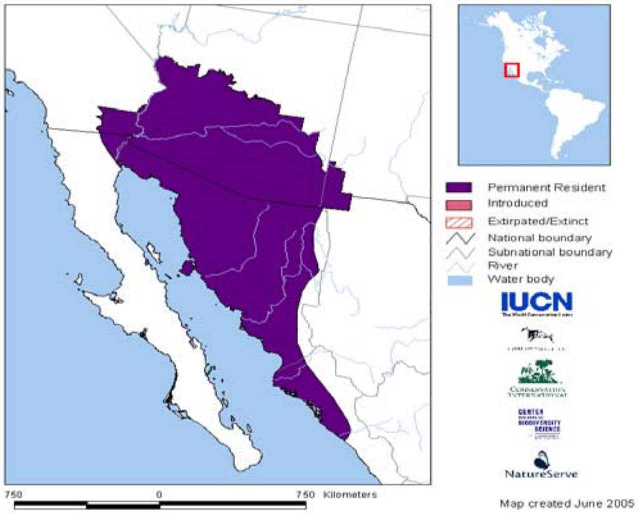
Current Range

Bufo alvarius is currently restricted to the Sonoran Desert in lowland and riparian areas of southern Arizona and adjacent corners of southeastern California, southwestern New Mexico and northeastern Baja California, through most of Sonora, and to seven miles west of Guamuchil, Sinoloa Mexico (Fouquette 1968, Fouquette 1970, Riemer 1955). This species is found at elevations ranging from above sea level to 1600 m (Cole 1962).

Total adult population size for this species is unknown (Natureserve 2006). *Bufo alvarius* is common throughout its range in Arizona but has declined in California and New Mexico (Natureserve 2006, Degenhardt et al. 1996, Jennings and Hayes 1994, Stebbins 1985). This species is believed to be extirpated from the California and the LCR region (Brennan and Holycross 2006, Jennings and Hayes 1994). The last sighting of this species in California was on 31 July 1955 (Jennings 1987 *in* Jennings and Hayes 1994). In 1980 and 1986, one individual was detected in dredge spoil and one individual was detected adjacent to a dredge spoil site located along the LCR in Arizona, 37 river km south of Blythe, California (Anderson and Ohmart 1982 *in* Jennings and Hayes 1994, J Rorabaugh pers. comm. *in* Jennings and Hayes 1994, Jennings and Hayes 1994). Five to 10 individuals were detected along agricultural borders on the Colorado Indian Reservation in the 1970s (B. Loudermilk, pers. comm. *in* Jennings and Hayes 1994). This species has not been recorded along the LCR since 1986. In 1991, surveys were conducted on the California side of the LCR Basin in the vicinity of Winterhaven, Palo Verde, Bard, Ferguson Lake, Goose Flats, and Cibola National Wildlife Refuge in which individuals of *Bufo alvarius* were not detected (King and Robbins 1991). Extensive surveys downstream of Imperial Dam during the 1980s and 1990s were conducted and *Bufo alvarius* was not detected (Rorabaugh pers. comm. *in* SAIC/Jones and Stokes 2003). A survey was conducted in August 1999 at three sites within Cibola National Wildlife Refuge (Mitchells Camp, Walters Camp, the Anderson and Ohmart Dredge Spoil Revegetation Site) and around Parker Dam (up to Black Meadow Landing). *Bufo alvarius* was not detected during these surveys (Rorabaugh pers. comm. *in* SAIC/Jones and Stokes 2003).

Three hybrids between *Bufo alvarius* and *Bufo woodhousii* were observed in Maricopa County, Arizona in 1959, 1995, and 1997 (Gergus et al. 1999). Secondary sexual characteristics, such as testes similar in shape to breeding males and advertisement calls, were present in these hybrids; however, reproductive competency of these individuals is unknown (Gergus et al. 1999). Fossil evidence suggests some genomic compatibility has been retained between *Bufo alvarius* and *Bufo woodhousii,* despite at least 6 million years of independent evolution (Gergus et al. 1999). Hybridization between *Bufo alvarius* and *Bufo woodhousii* may be due to the increase of females actively searching for males during the mating period (Gergus et al. 1999).

Figure 1: Range of *Bufo alvarius*.



Life History

General Description

The holotype of *Bufo alvarius* was described by Charles Girard from a specimen collected in Fort Yuma, Imperial County, California, in 1859. The lectotype was a female collected by Major G.H. Thomas in 1855 (Fouquette 1970). *Bufo alvarius* is a nocturnal toad in the family Bufonidae. Common names for this species are the Colorado River Toad and the Sonoran Desert Toad (Brennan and Holycross 2006). *Bufo alvarius* is a large anuran with snout-vent length (SVL) ranging from 110 to 187 mm. *Bufo alvarius* has leathery skin that ranges in color from olive brown to black with a few, low rounded tubercles and enlarged glands on the dorsal surfaces of the limbs (Fouquette 1970, Brennan and Holycross 2006). The length of each large paratoid gland equals the distance from nostril to tympanum; the width is less than half the length. Two distinct traits of this species are one to four conspicuous, whitish, rounded tubercle just behind the angle of the jaws, and distinct cranial crest curves above each eye (Fouquette 1970, Brennan and Holycross 2006). Females contain reddish-colored warts in straight lines on the dorsal surface. Males have larger nupitial pads on the thumbs than do females (Hill 1961).

Sullivan and Fernandez (1999) found SVL to be positively correlated with body mass for males. Snout-vent length and age, estimated by lines of arrested growth (LAG), were not significantly correlated in a population in Maricopa County, Arizona (Sullivan and Fernandez 1999). Growth rate appears to decrease with age (Sullivan and Fernandez 1999).

Tadpoles of this species have a brassy coloration, rounded tail, flattened body, and can reach a size of 57 mm total length (TL) (Deganhardt et al. 1996).

Breeding

Bufo alvarius breeds from May through August in ponds, slow moving streams, temporary pools or manmade structures that hold water (Stebbins 1985, Natureserve 2006). In Arizona, *Bufo alvarius* usually breeds in temporary pools formed by monsoon rains (Brennan and Holycross 2006). In a study conducted at flood control sites in north central Maricopa County, Arizona, rainfall greater than 25 mm within a 24-hour period was necessary for populations of *Bufo alvarius* to initiate breeding and chorusing activity (Sullivan and Fernandez 1999, Sullivan and Malmos 1994). Breeding and chorusing activity usually occurred one to three nights following rainfall events (Sullivan and Fernandez 1999, Sullivan and Malmos 1994). However, Fouquette (1970) and Arnold (1943) observed that while breeding activity is stimulated by rainfall, it is not necessary for reproductive activity. Sullivan and Fernandez (1999) observed the persistence of *Bufo alvarius* over a 6-year time period in the absence of successful breeding through many seasons.

Clutch size of *Bufo alvarius* is between 7,500 and 8,000 eggs per female. Eggs are 1.6 mm in diameter, 5-7 cm apart, and encased in a long single tube of jelly with a loose but distinct outline (Stebbins 1972 *in* NatureServe 2006, Wright and Wright 1949 *in* Fouquette et al. 2006). Eggs are deposited in shallow water of pools (Fouquette et al. 2005). Little is known about the length of development of eggs, larvae, or tadpole except that it takes less than 30 days for an egg to metamorphosis into a froglet (Musgrave and Cochran 1930, Brennan and Holycross 2006). When compared to other frogs and toads, *Bufo alvarius* develops from zygote to hatchling at a remarkably fast rate (Musgrave and Cochran 1930, Brennan and Holycross 2006). Eggs of *Bufo alvarius* have different characteristics than other members of the *Bufo boreas* group in completely lacking an inner gelatinous membrane and partitions between individual eggs (Savage and Schuierer 1961). Tadpoles are gray to golden brown and can reach up to 57 mm in length (Degenhardt et al. 1996).

Males and females reach sexual maturity at an SVL of 80-156 mm and 87-178 mm, respectively (Wright and Wright 1949 *in* Fouquette et al. 2005). Males utilize two strategies for pairing with females, including active searching and stationary calling from shallow water. The strategy chosen appears to be influenced by size of individual (Sullivan and Malmos 1994). Sullivan and Malmos (1994) noticed that males observed calling were significantly larger than those observed actively searching. When there are fewer males in a breeding aggregation, individuals call more frequently (Sullivan and Malmos 1994). Blair and Pettus (1954) observed that the call of *Bufo alvarius* does not play a role in breeding behavior; however, Sullivan and Malmos (1994) demonstrated in preliminary trials that females were attracted to the call of males (Sullivan and Malmos 1994). Further research on mate selection techniques needs to be conducted (Sullivan and Malmos 1994).

Bufo alvarius advertisement call consists of an emphasized note, followed by three progressively weaker notes. The fundamental frequency of the advertisement call is 1096 cycles per second and its duration is approximately 0.7 seconds in length (Blair and Pettus 1954). Sound pressure levels of advertisement calls were approximately 88 dB at about 0.5 m, similar to other bufonids (Sullivan and Malmos 1994). Release calls consist of a series of pulse groups lasting about 1 second. Advertisement and release calls differ in temporal structure (Sullivan and Malmos 1994). Release calls have a faster pulsation rate and a lower frequency than advertisement calls (Sullivan and Malmos 1994). Frequency or duration of advertisement or release calls is not related to body size or body temperature of individuals (Sullivan and Malmos 1994). Pulse rate of advertisement calls increase as body temperature of individual increases (Sullivan and Malmos 1994). Pulse rate of release calls decrease as body temperature of individual increases (Sullivan and Malmos 1994). *Bufo alvarius* arytenoids cartilages are believe to be activated only during production of release calls (Sullivan and Malmos 1994).

Diet

Bufo alvarius adults are active foragers and feed on invertebrates, lizards, small mammals, and amphibians (Brennan and Holycross 2006). Stomach content analysis of five *Bufo alvarius* specimens and intestinal analysis of one specimen produced members of the following orders, from most abundant to least abundant: *Coleoptera* (beetles), *Hymenoptera* (wasps, ants, and bees), *Isoptera* (termites), *Solpugida* (sun spiders), *Hemiptera* (true bugs), *Lepidoptera* (butterflies and moths), *Arachnida* (spiders, mites, scorpions), *Orthoptera* (grasshoppers, locusts, crickets), *Spirobolida* (millipedes), and *Scolopendromorpha* (centipedes) (Cole 1962). Tadpoles are believed to be algivorous and omnivorous. *Bufo alvarius* is able to eat prey that is protected by sting mechanisms or defensive secretions (Cole 1962).

Habitat

Bufo alvarius is a semi-aquatic toad that occurs primarily in desert habitat, including mesquite-creosote lowlands, but also inhabits arid grasslands, oak-woodland habitat, riparian areas, and pine-oak-juniper forest (Stebbins 1985, Fouquette 1970, Holycross et al. 1999). This species is found at elevations ranging from above sea level to 1610 m (Cole 1962, Stebbins 1985, Fouquette 1970). Holycross et al. *(*1999) observed this species in pine-oak woodlands, characterized by high densities of Chihuahua pine (*Pinus leiophylla*), Mexican pinyon, alligator bark juniper, and various oaks (*Quercus* spp.), and a native grassland in Chihuahuan desert scrub. Cole (1962) observed this species associated with *Agave, Ephedra, Prosopis, Slasola, Yucca, Gutierrezia*, and grasses. This species may have also expanded its range to agricultural areas when large-scale conversion of native habitat to agriculture took place.

Breeding habitat includes seasonal and permanent pools (Fouquette 1970, NatureServe 2006, Wright and Wright 1949 *in* Fouquette et al. 2005). MacMahon (1932 *in* Robbins and King 1991) describes preferred *Bufo alvarius* habitat as damp areas near permanent springs or manmade watering holes. They are also known to utilize artificial water bodies, such as canals, flood control impoundments, stock tanks, water irrigation ditches, and reservoirs (Gergus et al. 1999, Musgrave and Cochran 1930, Blair and Pettus 1954, Degenhardt et al. 1996). Blair and Pettus (1954) observed a breeding aggregation in a large stock tank. *Bufo alvarius* has been found inhabiting flood control sites at Adobe Dam and Cave Buttes and a cattle tank west of the Verde River in north-central Maricopa County, Arizona (Sullivan and Fernandez 1999, Sullivan and Malmos 1994). King and Robbins (1991) described agricultural drains, dam seepages, irrigation canals, and backwaters along the lower Colorado River as “marginal habitat” for this species.

Habitat for *Bufo alvarius* when dormant or refugium during active period includes subterranean shelters, such as rodent burrows, rock outcrops, or in hollows under watering troughs (Wright and Wright 1949 *in* Fouquette et al. 2005, Lowe 1964 *in* Fouquette et al. 2005, D. Beck unpublished data *in* Fouquette et al. 2005).

The general habitat preferences for *Bufo alvarius* are known but detailed information on habitat requirements and suitability is lacking (Jennings and Hayes 1994). Factors of habitat suitability include, but are not limited to, permanency of water sources, degree of water level fluctuation, water current, and quality of soil type for egg laying (King and Robbins 1991). Detailed data on habitat requirements are essential to understanding why this species is stable or thriving in southern Arizona but possibly extirpated from the lower Colorado Region and California.

Habitat creation projects implemented by the LCR MSCP may inadvertently provide breeding habitat for some species of frogs and toads. Habitat creation projects are flood irrigated and have irrigation structures in place. The Beal Lake Restoration project is irrigated by outlets in each field. Many of these outlets leak, creating small temporary or permanent ponds adjacent to the outlet. Pacific tree frogs (*Hyla regilla*) were observed breeding in these areas in April 2006. Other habitat creation projects may provide similar habitat to the Pacific tree frog and other species of frogs and toads.

Defense Behavior

Bufo alvarius is capable of emitting a poisonous substance (toxic indolealklamines) from its paratoid glands (Hanson and Vial 1956, Musgrave and Cochran 1930, Erspamer et al. 1967 *in* Fouquette et al. 2006, Cei et al. 1968 *in* Fouquette et al. 2006). The substance is discharged when the teeth of the predator sink into the granular glands (Hanson and Vial 1956). Toxins from this substance, when released in the air, had little effect on a striped skunk in a laboratory setting (*Mephitis mephitis*) and showed no effect on a domestic cat (*Felis domestica*) or a domestic dog (*Canis familiaris*). When the substance was directly released in a juvenile dog, the dog showed symptoms of increased drooling, lack of coordination, rapid breathing, convulsions, and lack of bladder control that lasted for approximately 50 minutes. Musgrave and Cochran (1930) reported a fox terrier dying after biting into *Bufo alvarius*. Musgrave and Cochran (1930) reported *Bufo alvarius* facial contact causing a police dog to be paralyzed for approximately 60 minutes. An effective defense posture of *Bufo alvarius* is to face the predator with its head, dorsum, and paratoid glands and make a hissing sound (Hanson and Vial 1956).

Biology

Bufo alvarius is sympatric with the spadefoot toad (*Scaphiopus* spp.), great plains toad (*Bufo cognatus*), red-footed toad (*Bufo punctatus*), and Woodhouse toad (*Bufo woodhoussii*) (Blair and Pettus 1954, Sullivan and Malmos 1994, Wright and Wright 1949 *in* Fouquette et al. 2005). The age of individuals of *Bufo alvarius* ranged from 2 to 4 years within a population at Adobe Dam in Maricopa County, Arizona (Sullivan and Fernandez 1999). Individuals of other species in the Bufonidae family have an average lifespan of 4 to 5 years (Fouquette et al. 2005).

Bufo alvarius migrates short distances between breeding and non-breeding habitats but no studies have been conducted on the length of these migrations (NatureServe 2006, Fouquette et al. 2005). Individuals may migrate several hundred meters from permanent to seasonal pools following heavy rains (Wright and Wright 1949 *in* Fouquette et al. 2005). Little research has been conducted dealing with the home range of *Bufo alvarius* (Fouquette et al. 2005).

There is no direct evidence of aestivation or torpor in this species; however, little research pertaining to this topic has been conducted. *Bufo alvarius* is dormant from September to April (Stebbins 1985).

Survey Methods

A variety of methods have been used to survey for amphibian species, including calling surveys, frogloggers, egg mass surveys, mark-recapture techniques, and visual encounter surveys (Jung et al. 2006, Droege 2006, Jung and Mitchell 2006, Muths 2006). Calling surveys provide trend and abundance estimates by multiplying the number of individuals heard by a calling index value (Droege 2006). Frogloggers are automated recording devices that are used in calling surveys to increase the frequency of data collected (Jung and Mitchell 2006). Accuracy of calling surveys and frog loggers are dependent on the ability of observers to identify calls made by amphibian species. Egg-mass surveys are used with pond and pool breeders. They provide estimates of abundance, reproductive outputs, and population trends (Jung et al. 2006). Visual encounter surveys, described by Crump and Scott (1994), Campbell and Christman (1982), and Corn and Bury (1990), are a widely used method that provides estimates of species richness, species list, and proportion of habitat occupied by target species (Howland et al. 1997, Muths 2006). Visual encounter surveys are effective in most habitats and for most species that breed in lentic water. They are conducted by observers walking though a designated area for a prescribed time, visually searching for animals (Muths 2006). The three standard sampling designs for visual encounter surveys are walk, transect, or quadrat designs (Muths 2006). Accurate and precise determination of leopard frog population size requires use of mark-recapture methods (Donnelly and Guyer 1994).

Threats

Habitat loss and alteration in the LCR region likely have had an impact on *Bufo alvarius* populations (Jennings and Hayes 1994). Extensive use of pesticides after World War II may have had an effect on this species (Jennings 1987a *in* Jennings and Hayes 1994). Nonnative species that have an effect on many native species, such as *Rana catesbieana* and *Apalone spinifera,* may also affect *Bufo alvarius* along the LCR. Illegal collection of *Bufo alvarius* for use in the drug trade is also a threat to this species (Weil and Davis 1994, Leavitt 1989 *in* Jennings and Hayes 1994).

Because detailed information is not available on habitat requirements of *Bufo alvarius*, the specific reasons why *Bufo alvarius* have declined along the LCR are unknown (Jennings and Hayes 1994). Furthermore, the historical abundance of the species in this region is not quantitatively known, so the extent of decline in this region can not be verified. Cole (1962 *in* Fouquette et al. 2005) reported *Bufo alvarius* as common near Tucson and west to the Colorado River. The reasons why this species is successful in agricultural and desert habitat in southern Arizona, but has shown severe decline along the LCR are unknown. In the case that extant populations of *Bufo alvarius* are discovered along the LCR or a translocation program is initiated, habitat requirements and factors negatively affecting populations of *Bufo alvarius* must be determined.

Predators include raccoons (*Procyon lotor),* possibly birds, other mammals, and reptiles (Wright 1966). Wright (1966) observed a raccoon ripping open the abdominal cavities on five *Bufo alvarius* adults and consuming the contents of the cavities but leaving the dorsal portion of the carcasses. Hanson and Vial (1956) observed that *Bufo alvarius* defensive posture and skin toxins protect it from the striped skunk (*Mephitis mephitis*). It is likely that adult *Bufo alvarius* are safe from most predators due to the toxicity of their paratoid secretions and their large size; however, no research study has been conducted on predators of *Bufo alvarius.*

Parasites known to affect *Bufo alvarius* are *Aplectana itzocanensis*, *Oswaldocruzia pipiens*, *Physaloptera* spp., *Physocephalus* spp., and *Rhabdias americanus* of the phyla Nemotoda and *Nematotaenia dispar* of the family Cestoidea (Goldberg et al. 1991).

Although populations of *Bufo alvarius* in southern Arizona appear to be thriving, they may be affected in the future, along with other species of desert toads, as rampant development in the areas next to Tucson and Phoenix convert agriculture and creosote flats into urban areas (Tom Jones pers. comm.).

Conservation Measures

CRTO1—Conduct research to better define the distribution, habitat requirements, and factors that are limiting the distribution of the Colorado River Toad.

A multiyear integrated research program is being developed and implemented to determine the range, status, habitat requirements, population biology, and factors that currently limit Colorado River Toad abundance and distribution, and factors that have contributed to the decline of the species in the LCR MSCP planning area.

CRTO2-Protect existing unprotected occupied Colorado River Toad habitat.
Based on results of research conducted under conservation measures CRTO1 and within funding constraints of the LCR MSCP, existing unprotected occupied Colorado River Toad habitat that is located through the research program will be protected.

CRTO3-Conduct research to determine feasibility of establishing the Colorado River Toad an unoccupied habitat.
Research necessary to determine the feasibility for successfully establishing the Colorado River Toad in unoccupied habitat will be conducted. If feasible, a pilot introduction into unoccupied habitat will be implemented, and the success of methods and establishment of the Colorado River Toad in unoccupied habitat will be monitored.

LCR MSCP Research and Monitoring Needs

Research and monitoring needs to fulfill LCR MSCP conservation measures for *Bufo alvarius* include:

1) Conduct system-wide surveys along the LCR to determine distribution and population status of the species within the LCR MSCP planning area. Surveys should take place before, during, or after summer thunderstorms (King and Robbins 1991).

2) Conduct microhabitat and macrohabitat studies to define breeding requirements for extant populations along the LCR (Jennings and Hayes 1994). Breeding requirements factors may include permanency of water sources, degree of water level fluctuation, water current, and quality of soil type for egg laying (King and Robbins 1991).

3) Conduct studies on population biology, such as determining migration from breeding to nonbreeding habitat, post-metamorphic migration patterns, and determining the home range of *Bufo alvarius.*

4) Determine factors that negatively affect extant populations or prohibit this species from occupying historical habitat along the LCR, including the effects of *Rana catesbieana* and *Apalone spinifera* on native populations of *Bufo alvarius.*

5) Determine the feasibility of establishing the Colorado River toad in unoccupied habitat, including conducting translocation and captive breeding studies. Research priorities pertaining to initiating a translocation program may include:

    a) Conduct a research study on conditions needed for *Bufo alvarius* to breed in captivity.

b) Conduct habitat evaluations across the historical range of *Bufo alvarius* along the LCR to define suitable habitat for relocations.
c) Conduct genetic studies across *Bufo alvarius* range to determine what source populations should be utilized for translocation.
d) Conduct research pertaining to egg mass collection and transportation, captive care and captive release, and other areas related to a successful translocation program in species of the Bufonidae family.
e) Establish a pilot translocation program and monitor the effectiveness of that program.

Literature Cited

AmphibiaWeb. 2006. Information on amphibian biology and conservation. Berkeley, California: AmphibiaWeb. Accessed: Aug 30, 2006. http://amphibiaweb.org/.

Arnold, L.W. 1943. Notes on two species of desert toads. Copeia 1943:128.

Blair, W.F., and D.I. Pettys. 1954. The mating call and its significance in the Colorado River Toad (*Bufo Alvarius* Girard). Texas Journal of Science 6:72-77.

Brattstrom, B.H. 1951. Two additions to the herpetofauna of Baja California, Mexico. Herpetological 7:196.

Brennan, T.C., and A.T. HolyCross. 2006. Amphibians and Reptiles in Arizona. Arizona Game and Fish Department, Phoenix.

Campbell, H.W., and S.P. Christman. 1982. Field techniques for herpetofaunal community analysis. Pages 193-200 *In:* N.J. Scott, Jr. (editor), Herpetological Communities. Wildlife Research Report 13. U.S. Department of the Interior, Fish and Wildlife Service.

Cole, C.J. 1962. Notes on the distribution and food habits of *Bufo alvarius* at the eastern edge of its range. Herpetological 18:172-175.

Cooper, J.G. 1869. The naturalist in California. American Naturalist 3:470-481.

Corn, P.S., and R.B. Bury. 1990. Sampling methods for terrestrial amphibians and reptiles. General Technical Report No. PNW-GTR-256. USDA Forest Service, Pacific Northwest Research Station, Portland. 34 p.

Crump, M.L., and N.J. Scott Jr. 1994. Standard techniques for inventory and monitoring-visual encounter surveys. Pages 84-92 *In:* W.R. Heyer, M.A. Donnelly, R.W. McDiarmid, L.C. Hayek, and M.S Foster (editors), Measuring and Monitoring Biological Diversity: Standard Methods for Amphibians. Smithsonian Institution Press, Washington, D.C.

Degandhardt, W.G., C.W. Painter, and A.H. Price. 1996. Amphibians and Reptiles of New Mexico. University of New Mexico Press, Albuquerque, New Mexico.

Donnelly, M.A., and C. Guyer. 1994. Estimating populations size- mark-recapture. Pages 183-200 *In:* W.R. Heyer, M.A. Donnelly, R.W. McDiarmid, L.C. Hayek, and M.S. Foster (editors), Measuring and Monitoring Biological Diversity: Standard Methods for Amphibians. Smithsonian Institution Press, Washington, D.C.

Droege, S. 2006. Amphibian Calling Surveys. Managers Monitoring Manual. U.S. Department of the Interior, U.S. Geological Survey, Patuxent Wildlife Research Center, Laurel, Maryland. Accessed July 27, 2006. http://www.pwrc.usgs.gov/monmanual.

Fouquette, M.J. 1968. Remarks on the type specimen of *Bufo avarius* Girard. The Great Basin Naturalist 28:70-72.

Fouquette, M.J. 1970. *Bufo alvarius.* Catalogue of American Amphibians and Reptiles 93.1-93.4.

Fouquette Jr., M.J, C.W. Painter, and P. Nanjappa. 2005. *Bufo alvarius* Cope. Colorado River Toad. *In:* Lanoo, M.J. (editor), Status and Conservation of U.S. Amphibians Volume 2: Species Accounts. University of California Press, Berkeley, California.

Gergus, W.A., K.B. Malmos, and B.K. Sullivan. 1999. Natural hybridization among distantly related toads (*Bufo alvarius, Bufo cognatus, and Bufo woodhousii*) in Central Arizona. Copeia 1999:281-286.

Goldberg, S.R., and C.E. Bursey. 1991. Helminths of three toads, *Bufo alvarius, Bufo cognatus* (Bufonidae), and *Scaphiopus couchii* (Pelonatidae), from Southern Arizona. Journal of the Helminthological Society of Washington 58:142-146.

Grinnell J., and C.L. Camp. 1917. A distributional list of the amphibians and reptiles of California. University of California Publication of Zoology 17:127-208.

Hanson, J.A., and J.L. Vial. 1956. Defensive behavior and effects of toxins in *Bufo alvarius.* Herpetologica 12:141-149.

Hill, W.J. 1961. The Developmental Pattern of the Colorado River Toad (*Bufo alvarius* Girard) From Stage 1-20. M.S. Thesis. Arizona State University, Tucson.

Holycross, A.T, B.G. Fodorko, and O. Fourie. 1999. Natural history notes: *Bufo alvarius* (Colorado River toad) habitat. Herpetological Review 20:90.

Jennings, M.R., and M.P. Hayes. 1994. Amphibian and reptile species of special concern in California. California Department of Fish and Game, Sacramento, California.

Jung R.E., L. Funk, and P. Nanjappa. 2006. Egg Mass Surveys. Managers Monitoring Manual. U.S. Department of the Interior, U.S. Geological Survey, Patuxent Wildlife Research Center, Laurel, Maryland. Accessed July 27, 2006. http://www.pwrc.usgs.gov/monmanual.

Jung R.E., and J.C. Mitchell. 2006. FrogLogger. Managers Monitoring Manual. U.S. Department of the Interior, U.S. Geological Survey, Patuxent Wildlife Research Center, Laurel, Maryland. Accessed July 27, 2006. http://www.pwrc.usgs.gov/monmanual.

Kauffeld, C.F. 1943. Field notes on some Arizona reptiles and amphibians. American Midland Naturalist 29:342-359.

King, T., and M. Robbins. 1991. A status of *Kinosternon sonoriens* and *Bufo alvarius* along the California side of the lower Colorado River Basin. Unpublished report prepared for the California Department of Fish and Game, Inland Fisheries Division, Region 5.

Lower Colorado River Multi-Species Conservation Program. 2004. *Lower Colorado River Multi-Species Conservation Program, Volume II: Habitat Conservation Plan.* Final. December 17. (J&S 00450.00) Sacramento, California.

Malkin, B. 1962. Seri ethnozoology. Occasional papers of Idaho State College Museum 7:1-68.

Mearns, Edgar Alexander. 1907. Mammals of the Mexican boundary of the United States. Part 1. Bulletin United States National Museum 56:1-530.

Musgrave, M.E., and D.M. Cochran. 1930. *Bufo alvarius* (Colorado River toad) Herpetological review 9:107.

Muths S. 2006. Visual Encounter Surveys for Amphibians. Managers Monitoring Manual. U.S. Department of the Interior, U.S. Geological Survey, Patuxent Wildlife Research Center, Laurel, Maryland. Accessed July 27, 2006. http://www.pwrc.usgs.gov/monmanual.

NatureServe. 2006. NatureServe Explorer: An online encyclopedia of life. Version 5.0 NatureServe, Arlington, Virginia. Accessed: August 1, 2006. http://www.natureserve.org/explorer.

Riemer, W.J. 1955. Comments on the distribution of certain Mexican toads. Herpetologica 11:17-23.

SAIC/Jones & Stokes. 2003. Lower Colorado River Multi-Species Conservation Program biological assessment. Preliminary Draft. May 22. (J&S 00-450.) Sacramento, California.

Savage J.M., and F.W. Schuierer. 1961. The eggs of toads of the *Bufo boreas* group, with descriptions of the eggs of *Bufo exsul* and *Bufo nelsoni.* Bulletin of Southern California Academy of Sciences. 60:93-99.

Slevin, J.R. 1928. The amphibians of western North America. Occasional Papers of California Academy of Sciences 16:1-152.

Stebbins. 1985. A Field Guide to Western Amphibians and Reptiles 2nd ed. Houghton Mifflin Co., Boston, Massachusetts. 336 p.

Storer, T.I. 1925. A synopsis of the Amphibia of California. University of California Publication of Zoology 27:1-342.

Sullivan, B.K., and K.B. Malmos. 1994. Call variation in the Colorado River toad (*Bufo Alvarius*): Behavioral and phylogenetic implications. Herpetologica 50:146-156.

Sullivan, B.K., and P.J. Fernandez. 1999. Breeding activity, estimated age-structure, and growth in Sonoran Desert Anurans. Herpetologica 55:334-343.

Vitt, L.J., and D. Ohmart. 1978. Herpetofauna of the lower Colorado River, USA: Davis Dam to the Mexican border. Proceedings of the Western Foundation of Vertebrate Zoology 2:35-72.

Weil, A.T., and W. Davis. 1994. *Bufo alvarius*: A potent hallucinogen of animal origin. Journal of Ethnopharmacology 41:1-8.

Wright, J.W. 1966. Predation on the Colorado River toad, *Bufo alvarius*, Herpetologica 22:127-128.

LOWLAND LEOPARD FROG
(*Rana yavapaiensis*)

Legal Status

The U.S. Forest Service, and the Bureau of Land Management have listed *Rana yavapaiensis* as a sensitive species (New Mexico Game and Fish 2004). *Rana yavapaiensis* is listed as an endangered species and is provided full protection in New Mexico (New Mexico Game and Fish 2004). *Rana yavapaiensis* is listed as a species of concern in Arizona (Arizona Game and Fish Department 2001). *Rana yavapaiensis* is extirpated from California (Vitt and Ohmart 1978) and extirpated from all but Hildago County in New Mexico (Scott 1992, Jennings 1995). *Rana yavapaiensis* is listed in the Special Protection Category by the Mexican government (Natureserve 2006). Natureserve ranks the status of *Rana yavapaiensis* as apparently secure on a global and national level (Natureserve 2006). Natureserve ranks the status of *Rana yavapaiensis* as apparently secure in the state of Arizona, presumed extinct from California, and critically imperiled in New Mexico (Natureserve 2006).

Distribution

Historical Range

Rana yavapaiensis was historically distributed along the lower Colorado River and tributaries in Nevada, California, Arizona, New Mexico, northern Sonora, and extreme northeast Baja California, Mexico, and from low elevation sites in the vicinity of the Grand Canyon, Arizona, and St. George, Utah, downstream to near the mouth of the Colorado River in Mexico (Jennings and Hayes 1994a, Jennings and Hayes 1994b, Vitt and Ohmart 1978). In California, the historical range extended discontinuously from San Felipe Creek near its junction with Carrizo Creek, and eastward through the Imperial Valley to the entire lower Colorado River (Jennings and Hayes 1994a, California Department of Fish and Game 2004). The Gila River formerly had suitable habitats that linked populations of the Colorado River and upper Gila River drainage together (Clarkson and Rorabaugh 1989). Historically, *Rana yavapaiensis* was found at 28 locations in California, 14 locations in New Mexico, and 302 locations in Arizona (Natureserve 2006).

Historical records for *Rana yavapaiensis* in Arizona have been recorded in Mohave, Yavapai, Coconino, La Paz, Maricopa, Gila, Pinal, Graham, Greenlee, Yuma, Santa Cruz, and Cochise counties. Historical records for *Rana yavapaiensis* in California have been recorded in San Bernardino, Riverside, and Imperial counties. Historical records for *Rana yavapaiensis* in New Mexico have been recorded in Catron, Grant, and Hildago counties (Vitt and Ohmart 1978, Clarkson and Rorabaugh 1989, Jennings and Hayes 1994b, Sredl et al. 1997).

Rana yavapaiensis was historically present along the lower Colorado River (LCR) and in its natural overflow lakes and tributary streams (Jennings and Fuller 2004, Vitt and Ohmart 1978, Clarkson and Rorabaugh 1989). Old records of specimens from above Hoover Dam indicate that at one time *Rana yavapaiensis* was widespread in the lower Colorado River (Vitt and Ohmart 1978). Stebbins (1951) found *Rana yavapaiensis* present below Imperial Diversion Dam. Grinnell and Camp (1917) reported individuals in Riverside County, California, along the Colorado River, north at least to Riverside Mountain. Slevin (1928) reported individuals in Yuma County, Arizona. Observations indicate that *Rana yavapaiensis* expanded its range in the Imperial Valley and along the Colorado River with the development of large-scale irrigated agriculture during the early part of the 20th century (Jennings and Fuller 2004).

Current Range

Rana yavapaiensis occurs in the southern half of Arizona as well as adjacent parts of Sonora, Mexico (Platz and Frost 1984). *Rana yavapaiensis* remains well represented in interior Arizona, south and west of the Mogollon Rim (Clarkson and Rorabaugh 1989, Sartorius and Rosen 2000). *Rana yavapaiensis* is present in every county in Arizona except Apache and Navaho, with 57% of all localities occurring in Gila, Maricopa, and Yavapai counties (Sredl et al. 1997b). Eighty percent of extant *Rana yavapaiensis* habitat is located in the Gila River drainage (Salt, Upper Gila, and Verde Agua Fria watersheds). Seventeen percent of extant habitat is located in the Bill Williams drainage and 2% of extant habitat is located in the headwaters of the Rio Concepcion and Rio Yaqui (Sredl et al. 1997b). Surveys for *Rana yavapaiensis* in Arizona in 1997 detected populations at 43 of 115 historical sites surveyed. Populations were detected at 61 sites where historical records for *Rana yavapaiensis* did not exist (Sredl et al. 1997b). *Rana yavapaiensis* occurs in several canyons in Saguaro National Park (Parker 2005). Populations of leopard frogs confirmed to be *Rana yavapaiensis* were discovered along the Colorado River in Surprise Canyon (west of Separation Canyon) in western Grand Canyon in the spring of 2004 (Gelciz and Drost 2004). This population was found 4 miles (6.4 km) up the canyon in a small pool (1 by 2 m) that had a sparse growth of cattails around the edge (Gelciz and Drost 2004). This is the first recent observation of *Rana yavapaiensis* in this location; however, this species was known to historically occur in this section of the river. This observation extends the current distribution of *Rana yavapaiensis* further north (Gelciz and Drost 2004). *Rana yavapaiensis* populations are declining in southeastern Arizona (AFGD 2001). Recent surveys by Vitt and Ohmart (1978), Clarkson and Rorabaugh (1989), and Jennings and Hayes (1994b) found extant populations of *Rana yavapaiensis* in only two localities in southwestern Arizona.

Recent surveys in California failed to detect *Rana yavapaiensis*; therefore, it is believed to be extirpated from California. The most recent record for *Rana yavapaiensis* in California was collected in 1965 from an irrigation ditch east of Calexico (Jennings and Hayes 1994b). *Rana yavapaiensis* in New Mexico is believed to be extirpated or present in very low numbers (Jennings 1995). A single individual was observed in Hidalgo county in 2000 (Sredl 2005). Leopard frogs in the Black Canyon (Colorado River) and Virgin River region in Nevada, once thought to be *Rana yavapaiensis,* were confirmed to be *Rana onca* (Jaeger et al. 2001).

Rana yavapaiensis is sympatric at intermediate elevations (1,180-1,700 m) with *Rana chiricahuensis* at a few sites in Central Arizona (Dillers Pond, Yavapai County), and in several canyon systems along the Arizona-Sonora borders in Santa Cruz County (Platz and Frost 1984). *Rana chiricahuensis* and *Rana yavapaiensis* hybrids are very rare (Platz and Frost 1984). Laboratory experimental crosses between *Rana chiricahuensis* and *Rana yavapaiensis* show interspecific genetic compatibility. Mating call characteristics appears be the premating reproductive isolative that accounts for the limited occurrence of hybrid individuals among these two species (Frost and Platz 1983). The inferior ability of species hybrids to form gametes may be a major post-mating reproductive isolating mechanism (Frost and Platz 1983).

Rana yavapaiensis had become very rare along the lower Colorado River by the early 1960s and was considered extirpated by 1974 (Vitt and Ohmart 1978, Clarkson and Rorabaugh 1989, Arizona Game and Fish Department 2001). *Rana yavapaiensis* was not found in Imperial Valley, California, the lower Colorado River, Arizona-California, and the lower Gila River, Arizona, from 1983 to 1987 (Clarkson and Rorabaugh 1989). *Rana yavapaiensis* is believed to be currently extirpated from the lower Gila and Colorado rivers in Arizona and adjacent California (Sredl et al. 1997b). *Rana yavapaiensis* has been recently reported from approximately 11.2 km (7 miles) upstream from the confluence of the Colorado River and the Bill Williams River, within the Bill Williams River NWR (Jennings and Hayes 1994b, Clarkson and Rorabaugh 1989, AGFD 1998 *in* SAIC/Jones & Stokes 2003).

Current Abundance

Mark-recapture studies conducted annually at six Arizona sites from 1991 to 1996 resulted in highly variable population estimates among sites and within sites, ranging from 19 to 1806 individuals per site (Sredl et al. 1997a). The Big Spring site, in Graham County, Arizona, was the only one of the six sites where monitoring was conducted for all 6 years. Population size was estimated at 313 individuals in 1991, 443 individuals in 1992, 156 individuals in 1993, 134 individuals in 1994, 92 individuals in 1995, and 70 individuals in 1996. Population size for Tule Creek, in Yavapai County, was estimated at 704 individuals in 1991, 887 individuals in 1992, and 1806 individuals in 1993. Population size for Barnhardt Mesa, in Gila County, was estimated at 863 individuals in 1994. Population size for Alamo Canyon, in Pima County, was estimated at 41 individuals in 1991 and 41 individuals in 1992. Population size for Horsefall Canyon, in Cochise County, was estimated at 59 individuals in 1994. Population size for Reed Spring, in Gila County, was estimated at 19 individuals in 1992. Population size for Thicket Spring, in Bloody Basin, Yavapai County, was estimated at 73 individuals in 1991 (Sredl et al. 1997a). For the Big Spring site, adult survivorship ranged from 0.06 to 1.72 and juvenile survivorship ranged from 0.03 to 1.83 from 1991 to 1996, with adult survivorship usually being higher than juvenile survivorship (Sredl et al. 1997a). For the Tule Creek Site, adult survivorship ranged from 0.04 to 3.92 and juvenile survivorship ranged from 0.08 to 2.09 from 1991 to 1996. (Sredl et al. 1997a). Survivorship seemed to follow a seasonal pattern, always being lowest in the winter (Sredl et al. 1997a).

Life History

General Description

J.E. Platz collected the first specimen of *Rana yavapaiensis* in 1971 from Tule Creek in Yavapai, Arizona. The specimen was a single adult male described by Platz and Frost in 1984 (Platz and Frost 1984, California Department of Fish and Game 2004). Common names for *Rana yavapaiensis* include the lowland leopard frog, San Felipe leopard frog, and the Yavapai leopard frog. *Rana yavapaiensis* is a species in the *Rana pipiens* complex that can be distinguished from other leopard frogs by the following characteristics: dorsolateral folds that are interrupted posteriorly and deflected medially in the sacral region, incomplete supralabial strip (diffuse anterior to eye), yellow pigmentation on the groin often extending onto the posterior venter and the underside of legs, lack of both vestigal oviducts and prominent external vocal sacs in males, and dark reticulate pattern on the posterior surface of thigh (Platz and Frost 1984, Platz 1988). The entire dorsolateral folds are prominently raised and light in color, in contrast to the dorsum background, which is grey-brown. Dorsal spots are dark brown. The posterior half of the dorsum displays faint longitudinal folds of the same color as the background of the dorsum. The exposed portion of the thighs bears prominent bars. The cloaca region has a blotched or reticulated pattern contrasting with a lighter background color. The greater portion of the posterior surface of the thighs bears a reticulate pattern of dark blotches on a cream background. The venter is cream in color and free of grey pigment, with the exception of the region of skin bordering the lower jaw (Platz and Frost 1984, Platz 1988). The mean snout-to-vent length (SVL) for males and females is 54.7 mm and 63.5 mm, respectively (Platz and Frost 1984, Platz 1988). *Rana yavapaiensis* is morphologically most similar to *Rana chiricahuensis* and *Rana magnaocularis* (Platz and Frost 1984). Based on chromosomal elements, *Rana magnaocularis* is the closest relative to *Rana yavapaiensis* (Platz and Frost 1984). *Rana yavapaiensis* and *Rana magnaocularis* are quantitatively similar genetically but are qualitatively dissimilar in developmental compatibility with other species (Platz and Frost 1984). Sceletochronology of *Rana yavapaiensis* indicates that individuals can live up to 3 years. Survivorship of adults and juveniles appear to be high in the spring and summer and lower in the fall and winter (Arizona Fish and Game Department 2001, Sredl et al. 1997a). Sredl et al. (1997a) detected a seasonal fluctuation in body size at two sites in Arizona; SVL was highest in frogs measured in April, lowest in June, and gradually increased through October. Males appear to grow faster than females (Sredl et al. 1997a).

Movement and Genetic Structure

Rana yavapaiensis populations occupying geothermal springs or springs at low elevations are likely active year round (Sredl 2005). There is little information on territories, aestivation, seasonal migrations, and torpor of *Rana yavapaiensis. Rana yavapaiensis* populations are primarily connected by movement through drainages and not along straight lines (Goldberg et al. 2004). Goldberg et al. (2004) studied populations of *Rana yavapaiensis* in Saguaro National Park and found distances between populations to range from 0.7 to 29.7 km. Populations of *Rana yavapaiensis* are more isolated from each other than other amphibians in Arizona (Goldberg et al. 2004). Goldberg et al. (2004) found that genetic differentiation was high and migration low among populations in different drainages in the Tucson Basin of southern Arizona. Thirty-four percent of the genetic diversity of all *Rana yavapaiensis* samples in the Tucson Basin can be attributed to variation among populations (Goldberg et al. 2004). Goldberg et al. (2004) concluded that populations of *Rana yavapaiensis* in Saguaro National Park are not going extinct and being recolonized from adjacent drainages, but instead adults are persisting at locations undetected for several years or recolonizing from unsampled locations further up drainages. Goldberg et al. (2004) also found that four of seven populations tested showed signs of a recent population bottleneck that has persisted through an estimated 17 generations. Population bottlenecks were likely caused by the drying up of most valley river systems in the area (Goldberg et al. 2004). Many extant populations of *Rana yavapaiensis* are small and isolated (Sartorius and Rosen 2000).

Benedict et al. (2004) studied *Rana yavapaiensis* metapopulation dynamics of 16 populations in the Bill Williams River drainage, which encompasses the Bill Williams River, Alamo Reservoir, and large portions of the Santa Maria and Big Sandy rivers. Data suggests that some gene flow occurs from the Big Sandy River to the Santa Maria River, but metapopulations in both drainages were functioning independently of each other (Benedict et al. 2004). Source populations were found in the high elevation reaches, upstream of the main channels, and were the most genetically distinct and unique (Benedict et al. 2004).

Breeding

Rana yavapaiensis egg masses are spheroidal and attached to vegetation, bedrock, or gravel. Egg masses develop into larvae in 15 to 18 days (Sartorius and Rosen 2000). Egg masses are found near the water surface (<2 cm depth) or are slightly emergent (Sartorius and Rosen 2000). Egg masses are deposited in both spring (March-May) and fall (September-October), with a distinct summer hiatus (Sartorius and Rosen 2000, Collins and Lewis 1979). Tadpoles metamorphose in the same year they were oviposited, or overwinter (Collins and Lewis 1979). Reproduction that occurs in the fall (September-October) often results in an overwintering population of larvae (Collins and Lewis 1979). Growth of tadpoles occurs in warm springs but is arrested in cold springs in other species of leopard frogs; this may occur in *Rana yavapaiensis* (Jennings 1990 *in* Sredl 2005, R, Jennings personal communication *in* Sredl 2005). Sartorius and Rosen (2000) observed that egg masses were primarily deposited in March; those that were deposited in late spring were about half the size of those deposited in March. Sartorius and Rosen (2000) observed that the majority of reproduction occurred in March through May and a much smaller amount of reproduction occurred from September to October. Winter breeding may occur in springs with warm water temperatures. Egg masses have been observed in January (Ruibal 1959, Collins and Lewis 1979, Frost and Platz 1983). Sartorius and Rosen (2000) found that the survivorship of egg masses is high but there is mortality among eggs of individual masses. The mortality usually occurs in the eggs that are near the top of the mass that were partially emergent and exposed to air (Sartorius and Rosen 2000). Sartorius and Rosen (2000) found that larval development and transformation occurred earlier in the season in dryer years than in wet years. Larvae metamorphose in 3 to 9 months (Arizona Game and Fish Department 2001). *Rana yavapaiensis* have an average SVL of 25-29 mm at metamorphosis (Platz 1988). Males reach sexual maturity when SVL measures 53.5 mm. The size of females when they reach sexual maturity is unknown (Arizona Game and Fish Department 2001). Sex ratios are generally 1:1 (Sredl et al. 1997).

Ruibal (1962) found that the temperature range of water for normal development of *Rana pipiens* (lowland form) is between 11°C and 29°C. *Rana pipien* egg masses (lowland forms) have not been found to be exposed to water temperatures greater than 25°C (Ruibal 1962). *Rana yavapaiensis* produces a mating call that comprises many notes (typically 6-16), with the first note 1.5-2 times in duration of repetitive segments. Internote duration is less than the note length. The internote time tends to decrease in length as the call sequence progresses (Platz and Frost 1984, Platz 1988) The pulse rate is relatively low (8 pulses per second at 20°C) and dominant frequency averages 1.8 khz (Platz and Frost 1984, Platz 1988). The pulse number per note varies, decreasing from approximately 11 pulses in the first note to 3-4 in the last of a series (Platz 1988). The mating call of *Rana yavapaiensis* is more similar to the mating call of *Rana magnaocularis* than to any other species of leopard frog (Platz and Frost 1984). Proximate cues that stimulate mating in *Rana yavapaiensis* are not well studied (Arizona Game and Fish Department 2001). Rainfall and water temperature have been mentioned as cues for other leopard frog species in the Southwest (Arizona Game and Fish Department 2001).

Habitat

Rana yavapaiensis occurs in ponds and stream pools along water systems in desert grasslands to pinyon juniper (Platz and Frost 1984). They occur at elevations ranging from sea level to 1817 m (Sredl et al. 1997). They are habitat generalists and breed in rivers, permanent streams, permanent pools in intermittent streams, beaver ponds, wetlands, springs, earthen cattle tanks, livestock drinkers, irrigation sloughs, wells, mine adits, and abandoned swimming pools (Platz and Frost 1984, Scott and Jennings 1985, Sredl and Saylor 1998 *in* Arizona Game and Fish Department 2001). Benedict (2002) detected *Rana yavapaiensis* occupying open water channels, higher elevation bedrock seeps, and an open cattle pond/spring in the Bill Williams Basin. *Rana yavapaiensis* occupied habitat in Arizona consists of 82% natural lotic habitats and 18% lentic habitats (primarily stock tanks) (Sredl et al. 1997). In lotic habitats, they are concentrated at springs, near debris piles, at heads of pools, and near deep pools associated with root masses (Jennings 1987 *in* Arizona Game and Fish Department 2001). Sartorius and Rosen (2000) documented *Rana yavapaiensis* using filamentous algae (*Cladophora*) mats for concealment. Habitat heterogeneity in the aquatic and terrestrial environment appears to be an important factor for *Rana yavapaiensis* (Arizona Game and Fish Department 2001). Shallow water and emergent and perimeter vegetation likely provide basking habitat. Deep water, root masses, undercut banks, and debris piles provide refuge from predators and potential hibernacual (Jennings 1987 *in* Arizona Game and Fish Department 2001, Platz 1988, Jennings and Hayes 1994A). Seim and Sredl (1994) found that juveniles were more frequently associated with small pools and marshy areas while adults were more frequently associated with large pools. Large pools are necessary for adult survival and reproductive efforts. Small pools and marshy habitats probably enhance juvenile survival (Seim and Sredl 1994). In semi-permanent aquatic systems, *Rana yavapaienis* may survive the loss of water by retreating into deep mud cracks, mammal burrows, or rock fissures (Howland et al. 1997).

Riparian overstory at extant *Rana yavapaiensis* localities in Arizona include cottonwoods (*Populus fremonti*), willows (*Salix* spp.), baccharis (*Baccharis glutinosa*), mesquite (*Prosopis* spp.), and saltcedar (*Tamarix* spp.). Marsh habitat at extant locations include three-square rushes (*Scirpus americanus*), spike rushes (*Eleocharis* spp.), narrow-leafed cattails (*Typha angustifolia*), and pondweed (*Potomageton* spp.) (Sredl et al. 1997a). *Rana yavapaiensis* populations in New Mexico are often associated with the Arizona sycamore (*Platanus wrightii*), seep-willow (*Baccharis glutinosa*), other trees and shrubs, and various forbs and graminoid plants (New Mexico Game and Fish 2004).

Populations of *Rana yavapaiensis* do not appear to be affected by the majority of flash flood events (Sredl et al. 1997a). Scouring floods may be beneficial to populations of *Rana yavapaiensis.* The Tule Creek site, in Arizona, became choked with vegetation, which eliminated open water habitats. A major scouring flood impacted the site and removed sediment and vegetation, which could have been attributed to the population size increase (Sredl et al. 1997a). Vegetation encroachment may have a negative effect on populations of *Rana yavapaiensis.* The University of Nevada Las Vegas is currently conducting research on the effects of vegetation encroachment on another species of leopard frogs that occur in the Colorado River (*Rana onca*). Preliminary results indicate that *Rana onca* strongly avoided segments of high vegetation cover, especially where *Scirpus* spp. is present.

Historically, along the LCR, *Rana yavapaiensis* inhabited slackwater aquatic habitats dominated by bulrushes, cattails, and riparian grasses near or under an overstory of cottonwoods and willows. *Rana yavapaiensis* were also observed in canals, roadside ditches, and ponds in the Imperial Valley as desert lands were converted to agriculture (Jennings and Hayes 1994a, Jennings and Hayes 1994b, Stebbins 1951).

Diet

Adults eat arthropods and other invertebrates (Stebbins 1985). Larvae are herbivorous and eat algae, organic debris, plant tissue, and minute organisms in water (Marti and Fisher 1998). Sartorius and Rosen (2000) found *Rana yavapaiensis* larvae feeding on filamentous algae (*Cladophora*) mats and the organisms within them. Other species of leopard frogs of the *Rana pipiens* complex feed on aquatic and terrestrial invertebrates (e.g., snails, spiders, and insects) and vertebrates (e.g., fish and other anurans) (Stebbins 1951). Research has not been conducted on feeding behavior or diet of *Rana yavapaiensis* adults or larvae (Sredl 2005).

Survey methods

A variety of methods have been used to survey for amphibian species, including calling surveys, frogloggers, egg mass surveys, mark-recapture techniques, and visual encounter surveys (Jung et al. 2006, Droege 2006, Jung and Mitchell 2006, Muths 2006). Calling surveys provide trend and abundance estimates; they are conducted by observers who record species heard, and results are adjusted by a calling index value (Droege 2006). Frogloggers are automated recording devices that are used in calling surveys to increase the frequency of data collected (Jung and Mitchell 2006). Calling surveys and frog loggers depend on the identification of calls made by amphibian species. Calling surveys are not the best option for ranids (leopard frogs). Leopard frogs call underwater and may rarely or never call above water. Calls that are above water tend to have a low noise level and are not easily heard (Droege 2006, Jung and Mitchell 2006). Egg-mass surveys are used with pond and pool breeders. They provide estimates of abundance, reproductive outputs and population trends (Jung et al. 2006). Visual encounter surveys, described by Crump and Scott (1994), Campbell and Christman (1982), and Corn and Bury (1990), are a widely used method that provides estimates of species richness, species list, and proportion of habitat occupied by target species (Howland et al. 1997, Muths 2006). Visual encounter surveys are effective in most habitats and for most species that breed in lentic water. They are conducted by observers walking though a designated area for a prescribed time, visually searching for animals (Muths 2006). The three standard sampling designs for visual encounter surveys are walk, transect, or quadrat designs (Muths 2006). Accurate and precise determination of leopard frog population size requires use of mark-recapture methods (Donnelly and Guyer 1994).

Sredl et al. (1997) used mark and recapture techniques to monitor populations of *Rana yavapaiensis.* Sartorius and Rosen (2000), Sredl et al. *(*1997b), and Clarkson and Rorabaugh (1989) used visual encounter surveys to monitor populations of *Rana yavapaiensis.* Sartorius and Rosen (2000) monitored egg masses of one *Rana yavapaiensis* population in a 2-km segment of Agua Caliente Canyon. Frosts and Platz (1983) monitored presence/absence of egg masses for the species of the *Rana pipiens* complex in the southwestern United States. The Arizona Game and Fish Department uses a standard riparian herpetological survey form for all riparian amphibian species that documents site-specific locality data, herpetofauna observations, and habitat conditions and characteristics (Sredl et al. 1997b). There is no standard *Rana yavapaiensis* survey protocol, but visual encounter and mark and recapture surveys are the most common techniques used with ranid species.

Threats

Rana yavapaiensis has been extirpated from more than 50% of its historical range and is believed to be extirpated from the LCR due to habitat loss, fragmentation, and introduction of nonnative species (Parker 2005, Clarkson and Rorabaugh 1989). Habitat has been lost due to conversion of desert habitat to agriculture, creation of large reservoirs that flooded historic habitat, and draining of wetlands (Clarkson and Rorabaugh 1989, Jennings and Hayes 1994). Damming, draining, and diverting of water have fragmented formerly contiguous aquatic habitat dispersal corridors that are necessary for establishment or maintenance of functional metapopulations (Natureserve 2006). Nonnative species establishment, in particular predatory fish, crayfish, and American bullfrog (*Rana catesbeiana),* in historical *Rana yavapaiensis* habitat, has been a major factor in the decline of the species (Clarkson and Rorabaugh 1989, Jennings and Hayes 1994b; Sredl et al. 1997b). Clarkson et al. (1986) concludes that the success of *Rana catesbeiana* and native fishes in the LCR is the likely cause for the disappearance of *Rana yavapaiensis* in this area. Sredl et al. (1997b) demonstrated a strong negative association between native ranids and nonnative predatory fishes. Predatory fishes, *Rana catesbeiana*, and crayfish block potential dispersal corridors between available aquatic habitats. The University of Nevada Las Vegas is conducting a study on the effects of nonnative predatory fish on another species of leopard frog (*Rana* onca) in the LCR. The study will determine whether introduced fish can be removed from a section of spring by use of fish barriers and common eradication techniques, and determine whether the removal of fish increased egg and tadpole presence and metamorph-juvenile frog recruitment (Jaeger et al. 2004). Other factors that have contributed to the decline of *Rana yavapaiensis* along the LCR are the loss of cottonwood-willow habitat, increased salinity levels of aquatic habitat, fire, water pollution, increased levels of incident ultraviolet radiation, heavy grazing, invasion of saltcedar, drought, and disease (Clarkson and Rorabaugh 1989, Jennings and Hayes 1994b, Sredl et al. 1997b).

Rana yavapaiensis populations in the San Felipe Creek drainage were eliminated by flooding and increased salinity levels (Jennings and Hayes 1994). Ruibal (1959) found that salinities greater than 5 0/00 (parts per thousand) were lethal to developing eggs in *Rana pipiens* (lowland form). Salinities ranging from 3.8 to 4.6 0/00 were semi-lethal to developing eggs. Salinities greater than 2.5 0/00 always caused some defect or abnormality in developing eggs (Ruibal 1959). The lethal minimum salt concentration tolerance of adult *Rana pipiens* (lowland form) is between 6 0/00 and 13 0/00 (Ruibal 1959).

The previously mentioned factors that have contributed to the decline of *Rana yavapaiensis* disrupted metapopulation dynamics (groups of individuals inhabiting a system of habitat patches connected by migration across contiguous habitat) of leopard frogs. Large aquatic habitats are dominated by nonnative species. Native leopard frog populations are reduced to small, isolated pockets of habitat that only support small, unstable populations. Large core populations no longer exist. Dispersal corridors between populations either no longer exist or are blocked by nonnative species (Sredl et al. 1997b). The low connectivity of *Rana yavapaiensis* populations suggest that this species is unlikely to recolonize sites when populations are extirpated (Goldberg et al. 2004).

Rana berlandieri was introduced into the LCR near Yuma, Arizona, from either Texas or New Mexico, between 1965 and 1971. *Rana berlandieri* was probably transported from the Imperial Valley Irrigation District Fish Hatchery through the Dogwood Canal, Central Main Canal, and the All American Canal to the Imperial Valley (Jennings and Hayes 1994b). Since 1981, *Rana berlandieri* has expanded its range west into Imperial Valley and south along the Rio Colorado, but has yet to expand its range north along the Colorado River (Clarkson and Rorabaugh 1989, Rorabaugh et al. 2002). *Rana berlandieri* has been collected from more than 53 sites in the lower Gila and Colorado river valleys; 21 of those sites are along the LCR near Yuma, Arizona, and in the Imperial Valley (Jennings and Hayes 1994, Platz et al. 1990, Rorabaugh et al. 2002). This species invades new habitats by dispersal via rivers, agricultural areas, ditches, and canals, and introduction by humans (Rorabaugh et al. 2002). *Rana berlandieri* appear to coexist with *Rana catesbeiana* and on occasion, replace them (Jennings and Hayes 1994). *Rana yavapaiensis* was eliminated from the LCR before the introduction of *Rana berlandieri;* therefore, *Rana berlandieri* has not appeared to be a factor in the extirpation of *Rana yavapaiensis* from the LCR (Jennings and Hayes 1994). The presence of *Rana berlandieri* may prevent recolonization of *Rana yavapaiensis* along the LCR but there are no data on the effects of *Rana berlandieri* on native fauna (Natureserve 2006, Platz et al. 1990). *Rana berlandieri* is a large leopard frog, and in Texas, stomach contents of adult specimens frequently contained small leopard frogs. Larger species of leopard frogs are capable of producing larger egg masses, possibly out-competing smaller species of leopard frogs (Platz et al. 1990). Smaller species of leopard frogs, such as *Rana yavapaiensis,* may suffer both in terms of predation and reproductive competition from *Rana berlandieri* (Platz 1990).

Rana catesbeiana is an introduced species in the Southwest that may have an effect on native leopard frog populations. During the summer of 1981, it was detected at an average density of 9.1 per linear kilometer in the lower Colorado River between Laguna and Morelos dams, Arizona-California (Clarkson and DeVos Jr. 1986). *Rana catesbeiana* appeared to be significant predators of *Rana yavapaiensis* when it was declining in southeastern Arizona (New Mexico Game and Fish 2004). Moyle (1973) cited *Rana catesbeiana* as the single most important factor in the elimination of *Rana aurora* from the San Joaquin Valley. Schwaibe and Rosen (1988) concluded that *Rana catesbeiana* was one of the reasons why populations of *Rana yavapaiensis* and other species of leopard frogs were decreasing on the San Bernardino National Wildlife Refuge. Hayes and Jennings (1986) conclude that existing data does not support the hypothesis that *Rana catesbeiana* are the most important agent in ranid species decline, and that other causes, such as nonnative fish and habitat alteration, are equally responsible. Data on the precise timing of habitat modification relative to the introduction and establishment of bullfrogs and other exotic predators is not available, and thus, cannot provide insights into which factor was most significant (Jennings and Hayes 1994). Preliminary data from bullfrog removal experiments are inconclusive as to whether bullfrog control measures may augment recruitment in *Rana yavapaiensis* and other species (Schwaibe and Rosen 1988). Sartorius and Rosen (2000) found that *Rana catesbeiana* has appeared to replace *Rana yavapaiensis* in modified habitats (reservoirs, large deep stock ponds, and other impoundments) in its historical range in Arizona. *Rana catesbeiana* appears to be absent in native lotic habitats where extant populations of *Rana yavapaiensis* occur in central Arizona (Sartorius and Rosen 2000, Clarkson and Rorabaugh 1989).

Fire is known to have an impact on populations of *Rana yavapaiensis.* The Box Canyon fire in Saguaro National Park caused large amounts of ash, gravel, and course sand to be carried to ephemeral stream channels. Within 3 years after the fire, all but a few of the 32 pools in Loma Verde Wash, where *Rana yavapaiensis* was previously found, were buried in sediment and remained buried as of the summer of 2005 (Parker 2005). A large pool in the Wildhorse Canyon Wash, in Saguaro National Park, has remained buried for 16 years after the Chiva fire (Parker 2005). The United States Geological Service, in cooperation with the National Park Service, is conducting a study of hydrologic changes caused by uncontrolled wildfires and the effects of increased sediment transport and deposition on leopard frog habitat in the Rincon District of Saguaro National Park (Parker 2005). Objectives of the 3-year project include estimating the background rates of sedimentation in perennial bedrock pools, determining mechanisms of sediment delivery from burned areas, determining the change in sediment yields caused by burning of watersheds, determining source areas of excess sedimentation in burned areas and their physical characteristics, and estimating potential sediment yield from unburned areas in the event of future uncontrolled fires (Parker 2005).

Chrytidiomycosis is a cutaneous infection of wild frogs and toads caused by the fungal agent *Batrachochytrium dendrobatidis.* Chrytidiomycosis was found to be the cause of death in 29 *Rana yavapaiensis,* 2 *Rana chiricahuensis,* and 2 *Hyla arenicolor* collected at eight locations in southern, central, and eastern Arizona. Frogs were collected during December of 1992, October-February of 1997-1998, and December-February of 1998-1999 (Bradley et al. 2002). Lesions found on the frogs were consistent with chrytidiomycosis, and included diffuse reddening of the skin of the abdomen, pelvic area, and legs. Microscopic lesions included epidermal hyperplasia, hyperkeratosis, and colonization of the keratinized layers of the epidermis sporangia of the chytrid (Bradley et al. 2002). Preliminary laboratory data show that *Rana yavapaiensis* experiences only sporadic mortality when exposed to *Batrachochytrium dendrobatidis* in the laboratory. Richards (2004) found that the growth rate of frogs not exposed to the bacteria was not significantly different than the growth rate of frogs exposed to the bacteria in the laboratory. Neither the frogs that were exposed to the bacteria and those not exposed showed any signs of morbidity or infection. Davidson et al. (2003) found that mortality of *Rana boylii* and *Rana yavapaiensis* was sporadic and unrelated to dose or strain of chytrid bacteria. Die offs in the wild may be a combination of chrytidiomycosis and other factors such as habitat loss, pesticides, nonnative predators, drought, temperature, and/or stress (Richards 2004, Davidson et al. 2003). Outbreaks of bacterial infections, including chrytidiomycosis and red-leg, can be caused by low air temperatures and overcrowding. Sredl et al. (1997a) documented two occasions where *Rana yavapaiensis* populations were stressed due to drought and low temperatures, which brought on a bacterial infection (red-leg) that reduced population size dramatically.

Predators of *Rana yavapaiensis* tadpoles are suspected to be insects (belostomatids, notonectids, dytiscids, anisopterans), vertebrates (native and nonnative fish, tiger salamanders, garter snakes [*Thamnophis* spp.]), mud turtles (*Kinosternon sonoriense*), great blue herons (*Ardea herodias*), and other birds. Predators of juvenile and adult frogs are suspected to be native and nonnative fish, American bullfrogs, mud turtles, garter snakes, great blue herons, black hawks (*Buteogallus anthracinus*), and mammals (rats, coyotes, gray foxes, raccoons, ringtail cats, coatis, black bears, badgers, skunks, bobcats, and mountain lions) (Sredl et al. 1997a). There have been no detailed research studies dealing with predators of *Rana yavapaiensis* (Sredl et al. 1997a). Large adults likely eat juvenile frogs or large larvae, but no research studies on this question has been conducted (Sredl 2005).

LCR MSCP Conservation Measures

LLFR1—Conduct research to better define distribution, habitat requirements, and factors that are limiting the distribution of the lowland leopard frog.
A multiyear integrated research program will be developed and implemented to determine the range, status, habitat requirements, population biology of lowland leopard frogs—factors that currently limit abundance and distribution—and factors that have contributed to the decline of the species in the LCR MSCP planning area.

LLFR2—Protect existing unprotected occupied lowland leopard frog habitat.
Based on results of research conducted under conservation measures LLFRO1 and within funding constraints of LCR MSCP, existing unprotected occupied lowland leopard frog habitat that is located through the research program will be protected.

LLFR3—Conduct research to determine feasibility of establishing the lowland leopard frog in unoccupied habitat.
Research necessary to determine the feasibility for successfully establishing the lowland leopard frog in unoccupied habitat will be conducted. If feasible, a pilot introduction into unoccupied habitat will be implemented, and the success of methods and establishment of the lowland leopard frog in unoccupied habitat will be monitored.

LCR MSCP Research and Monitoring Needs

Research and monitoring needs to fulfill LCR MSCP conservation measures for *Rana yavapaiensis* include:

1) System-wide surveys along the LCR to determine distribution and population status. *Rana yavapaiensis* is believed to be extirpated from the LCR (Vitt and Ohmart 1978, Clarkson and Rorabaugh 1989, Arizona Game and Fish Department 2001). However, surveys conducted along the LCR, in Arizona and California, were either of a general nature or limited in their scope (Jennings and Hayes 1994a). Surveys conducted along the LCR for *Rana yavapaiensis* were part of larger projects, such as state-wide surveys for leopard frogs or amphibians (Clarkson and Rorabaugh 1989, Sredl et al. 1997, Jennings and Fuller 2004). Intensive presence/absence surveys, specifically targeting *Rana yavapaiensis* and repeated over several years at historical and potential habitat within its historic range, need to be conducted along the LCR to ascertain the species' current status in this area (Jennings and Hayes 1994b). Visual encounter surveys should be conducted, with multiple visits to each site (Sredl et al. 1997). In the event that extant populations are found, mark and recapture studies should be conducted to accurately determine the size of the population. If extant populations are found, water temperature and water quality data, specifically salinity, should be collected in occupied habitat.

The University of Nevada-Las Vegas is currently conducting leopard frog surveys in the Colorado River and tributary drainages, upstream from the Virgin River, in the western portion of the Colorado River Basin. One of the objectives of the surveys is to determine the distribution of *Rana yavapaiensis* populations (Jaeger and Riddle 2005). The data gathered from this project can be used to supplement data gathered from system-wide surveys along the LCR.

2) Conduct studies to define habitat requirements. Record macro- and micro-habitats in areas where extant populations of *Rana yavapaiensis* occur to specifically define the importance of habitat heterogeneity to this species. The role of habitat heterogeneity within the aquatic terrestrial environment is unknown but likely important (Sredl 2005). These studies would quantitatively define preferable habitat characteristics for *Rana yavapaiensis*, especially those related to habitat heterogeneity. Dispersal corridor habitat, winter habitat use, and effects of vegetation encroachment also needed to be determined.

3) Determine factors that limit lowland leopard frog distribution and abundance, including defining the effects of *Rana berlandieri* on *Rana yavapaiensis;* determine if the bacteria *Batrachochytrium dendrobatidis*, which causes chrytidiomycosis, causes mortality in *Rana yavapaiensis* independently or if cofactors are needed for mortality to occur (Richards 2004); determine the effects of nonnative predatory fish and crayfish on *Rana yavapaiensis* populations; and determine the relationship between riparian plant species composition and population declines in *Rana yavapaiensis*. Some research has been done, or is currently being conducted, on factors that negatively affect *Rana yavapaiensis* or are preventing recolonization of this species along the LCR.

4) Determine the potential for establishing lowland leopard frog in unoccupied habitat. The Clark County LCR MHSCP has a successful translocation program with *Rana onca*, another species of leopard frog that occurs along the LCR (RLFCT 2005). Techniques and information gained from that program, pertaining to egg mass collection and transportation, captive care, and captive release, can be used with a *Rana yavapaiensis* translocation program. For translocation programs, source populations to replace extirpated populations should be the nearest population as measured along drainages (Goldberg et al. 2004). The first research priority is to conduct a study on conditions needed for *Rana yavapaiensis* to breed in captivity. The second research priority is to conduct habitat evaluations across the historical range of *Rana yavapaiensis* along the LCR to define suitable habitat for relocations. Water temperature should be between 11°C to 29°C and salinity levels should be below 2.5 0/00 (Ruibal 1959, 1972). Habitat evaluations should include suitable breeding habitat and suitable dispersal corridors. Nonnative species removal or habitat restoration may have to take place in some localities before translocation can occur. The third research priority would be to establish a pilot translocation program and monitor the effectiveness of that program.

5) Conduct studies on population biology, such as prey base, home range, and seasonal migration.

Literature Cited

Arizona Game and Fish Department. 2001. *Rana yavapaiensis.* Unpublished abstract compiled and edited by the Heritage Data, Management System, Arizona Game and Fish Department, Phoenix. 6 p.

Benedict, N. 2002. Molecular evaluation of the metapopulation structure of lowland leopard frogs (*Rana yavapaiensis*) in the Bill Williams River drainage, of western Arizona. Arizona Game and Fish Department Heritage Grant #I98026. University of Denver, Department of Biological Sciences, Denver, Colorado.

Bradley, G.A., P.C. Rosen, M.J Sredl, T.R. Jones, and J.E. Longcore. 2002. Chytridiomycosis in native Arizona frogs. Journal of Wildlife Diseases 38:206-212.

California Department of Fish and Game. 1994. Amphibian and Reptile Species of Special Concern in California, Lowland Leopard Frog. Sacramento.

Campbell, H.W., and S.P. Christman. 1982. Field techniques for herpetofaunal community analysis. Pages 193-200 *In:* N.J. Scott, Jr. (editor), Herpetological Communities. U.S. Department of the Interior, Fish and Wildlife Service, Wildlife Research Report 13.

Clarkson R.W., and J.C Devos Jr. 1986. The bullfrog *Rana catesbeiana* in the lower Colorado River, Arizona-California Journal of Herpetology 20:42-49.

Clarkson, R.W., J.C. Rorabaugh, and J.E. Platz. 1986. Status and future of ranid frogs in Arizona. Proceedings of Arizona Nevada Academy of Science 21:21.

Clarkson, R.W., and J.C. Rorabaugh. 1989. Status of leopard frogs (*Rana pipiens* complex: Ranidae) in Arizona and Southeastern California. Southwestern Naturalist 34:531-538.

Collins, J.P., and M.A. Lewis. 1979. Overwintering tadpoles and breeding season variation in the Rana Pipiens Complex in Arizona. Southwestern Naturalist 24: 371-396.

Corn, P.S., and R.B. Bury. 1990. Sampling methods for terrestrial amphibians and reptiles. General Technical Report No. PNW-GTR-256. USDA Forest Service, Pacific Northwest Research Station, Portland. 34 p.

Crump, M.L., and N.J. Scott Jr. 1994. Standard techniques for inventory and monitoring-visual encounter surveys. Pages 84-92 *In:* W.R. Heyer, M.A. Donnelly, R.W. McDiarmid, L.C. Hayek, M.S Foster (editors), Measuring and Monitoring Biological Diversity: Standard Methods for Amphibians. Smithsonian Institution Press, Washington, D.C.

Davidson E.W., M. Parris, J.P. Collins, J.E. Longcore, A.P. Pessier, and J.B Brunner. 2003. Pathogenicity and transmission of Chrytidiomycosis in Tiger Salamanders (*Ambystoma tigrinum*).

Donnelly, M.A., and C. Guyer. 1994. Estimating populations size-mark-recapture. Pages 183-200 *In:* W.R. Heyer, M.A. Donnelly, R.W. McDiarmid, L.C. Hayek, M.S. Foster (editors), Measuring and Monitoring Biological Diversity: Standard Methods for Amphibians. Smithsonian Institution Press, Washington, DC.

Droege S. 2006. Amphibian Calling Surveys. Managers Monitoring Manual. U.S. Department of the Interior, U.S. Geological Survey, Patuxent Wildlife Research Center, Laurel, Maryland. Accessed July 27, 2006. http://www.pwrc.usgs.gov/monmanual.

Frost, J.S, and J.E. Platz. 1983. Comparative assessment of modes of reproductive isolation among four species of leopard frogs (*Rana pipiens* complex). Evolution 37:66-78.

Gelcis, L., and C. Drost. 2004. A surprise in Surprise Canyon: A new amphibian species for the Grand Canyon National Park. Nature Notes Grand Canyon National Park News and Information. Fall 2004.

Goldberg, S.R., C.R. Bursey, and H. Cheam. 1998. Helminths of two native frog species (*Rana chiricahuensis, Rana yavapaiensis*) and one introduced frog species (*Rana catesbieana*) (Ranidae) from Arizona.

Goldberg, C., D.E. Swann, and J.E. Wallace. 2004. Genetic Structure of Lowland Leopard Frog (*Rana yavapaiensis*) Populations in Southern Arizona. Final Report. Western National Parks Association (Grant 03-11) and Saguaro National Park.

Goldberg, C.S., D.E. Swann, and J.E. Wallace. 2005. Genetic structure of remaining lowland leopard frog populations in southeastern Arizona. Abstract given at the joint meeting of ichthyologists and herpetologists. Tampa, Florida.

Grinnell, J., and C.L. Camp. 1917. A distributional list of the amphibians and reptiles of California. University of California Publication of Zoology 17:127-208.

Hayes, M.P., and M.R Jennings. 1986. Decline of ranid frog species in western North America: Are bullfrogs (*Rana catesbeiana*) responsible? Journal of Herpetology 20:490-509.

Howland, J.M., M.J. Sredl, and J.E. Wallace. 1997. Validation of visual encounter surveys. Pages 21-36 *In:* M.J Sredl (editor), Ranid frog conservation and management. Nongame and Endangered Wildlife Program Technical Report 121. Arizona Game and Fish Department, Phoenix, Arizona.

Jaeger, J.R., B.R. Riddle, R.D. Jennings, and D.F. Bradford 2001. Rediscovering *Rana onca*: Evidence for phylogenetically distinct leopard frogs from the border region of Nevada, Utah, and Arizona. Copeia:339-354.

Jaeger, J., B. Riddle, and R. Haley. 2004. Evaluation of the impact of vegetation encroachment on relict leopard frog populations. University of Nevada Las Vegas and National Park Service. Clark County MSHCP Project. Last edited 4/15/2004.

Jaeger, J., and B. Riddle. 2005. Evaluation of experimental habitat manipulations on relict leopard frog populations. University of Nevada Las Vegas. Clark County MSHCP Proposal. Last edited on 04/04/2005.

Jaeger, J., and B. Riddle. 2005. Delineation of distribution, evaluation of relatedness, and assessment of connectivity for leopard frog populations (*Rana* spp.) within the management zone of the relict leopard frog (*Rana onca*). University of Nevada Las Vegas. Clark County MSHCP Proposal. Last edited on 5/05/2005.

Jennings, M.R., and M.P. Hayes. 1994a. Amphibian and reptile species of special concern in California. California Department of Fish and Game, Sacramento, California.

Jennings, M.R., and M.P. Hayes. 1994b. Decline of native ranid frogs in the Desert Southwest. Pages 183-211 *In:* P.R. Brown and J.W Wright (editors), Herpetology of the North American deserts, proceedings of a symposium. Special Publication 5. Southwestern Herpetologists Society. Serpent's Tale Books, Excelsior, Minnesota.

Jennings, M.R., and M.M. Fuller. 2004. Origin and distribution of leopard frogs, *Rana pipiens* complex, in California. California Fish and Game 90:119-139

Moyle, P.B. 1973. Effects of introduced bullfrogs, *Rana catesbeiana,* on the native frogs of the San Joaquin Valley, California. Copeia 1973:18-22

Jung R.E., L. Funk, and P. Nanjappa. 2006. Egg Mass Surveys. Managers Monitoring Manual. U.S. Department of the Interior, U.S. Geological Survey, Patuxent Wildlife Research Center, Laurel, Maryland. Accessed July 27, 2006. http://www.pwrc.usgs.gov/monmanual.

Jung, R.E., and J.C. Mitchell. 2006. FrogLogger. Managers Monitoring Manual. U.S. Department of the Interior, U.S. Geological Survey, Patuxent Wildlife Research Center, Laurel, MD. Accessed July 27, 2006. http://www.pwrc.usgs.gov/monmanual.

Lower Colorado River Multi-Species Conservation Program. 2004. Lower Colorado River Multi-Species Conservation Program, Volume II: Habitat Conservation Plan. Final. December 17. (J&S 00450.00.) Sacramento, California.

Muths, S. 2006. Visual Encounter Surveys for Amphibians. Managers Monitoring Manual. U.S. Department of the Interior, U.S. Geological Survey, Patuxent Wildlife Research Center, Laurel, Maryland. Accessed July 27, 2006. http://www.pwrc.usgs.gov/monmanual.

Natureserve. 2006. NatureServe Explorer: An online encyclopedia of life. Version 4.7. Natureserve, Arlington, Virginia. Accessed: June 29, 2006. http://www.natureserve.org/explorer.

New Mexico Game and Fish Department. 2004. BISON species account 20030. Biota Information System of New Mexico BISON. Accessed: July 17, 2006. http://fwie.fw.vt.edu

Parker, J.T.C. 2005. Effects of post-wildfire sedimentation on leopard frog habitat in Saguaro National Park. Fact Sheet 2005-3140. U.S. Geological Survey. Tucson, Arizona.

Platz, J.E. 1988. *Rana yavapaiensis*—Catalogue of American Amphibians and Reptiles 418:418.1-418.2.

Platz, J.E., and A.L. Platz 1973. *Rana pipiens* complex hemoglobin phenotypes of sympatric and allopatric populations in Arizona. Science 179:1334-1336.

Platz, J.E., and J.S. Frost. 1984. *Rana yavapaiensis,* a new species of leopard frog (*Rana pipiens* complex). Copeia 1984:940-948.

Platz, J.E., R. Clarkson, J.C. Rorabaugh, and D.M. Hillis. 1990. *Rana berlandieri*: Recently introduced populations in Arizona and southeastern California. Copeia: 324-333.

Relict Leopard Frog Conservation Team. 2005. Conservation agreement and rangewide conservation assessment and strategy for the relict leopard frog (*Rana onca*).

Richards, K.L. 2004. The effect of a fungal pathogen, *Batrachochytrium dendrobatidis,* on lowland leopard frogs (*Rana yavapaiensis*). Honors Dissertation. School of Life Sciences, Arizona State University, Tempe, Arizona.

Richards, K.L., J.B. Brunner, V. Miera, E.W. Davidson, and J.P. Collins. 2004. The effect of a fungal pathogen, *Batrachochytrium dendrobatidis,* on lowland leopard frogs (*Rana yavapaiensis*). Poster presentation at undergraduate research symposium. School of Life sciences, Arizona State University.

Rorabaugh, J.C., M.J. Sredl, V. Miera, and C.A. Drost. 2002. Continued invasion by an introduced frog (*Rana berlandieri),* southwestern Arizona, southeastern California and Rio Colorado, Mexico. Southwestern Naturalists 47:12-20.

Ruibal, R. 1959. The ecology of a brackish water population of *Rana pipiens*. Copeia 1959: 315-322.

Ruibal, R. 1962. The ecology and genetics of a desert population of *Rana pipiens.* Copeia 1962:189-195.

SAIC/Jones & Stokes. 2003. Lower Colorado River Multi-Species Conservation Program biological assessment. Preliminary Draft. May 22. (J&S 00-45-). Sacramento, California.

Sartorius, S.S., and P.C. Rosen 2000. Breeding phenology of the lowland leopard frog (*Rana yavapaiensis*): Implications for conservation and ecology. The Southwestern Naturalist 45:267-273.

Seim, S.G., and M.J. Sredl. 1994. Lowland leopard frog micro-habitat use in Arizona. Annual Meeting of Arizona and New Mexico Chapters of the Wildlife Society, Sierra Vista, Arizona.

Slevin, J.R. 1928. The amphibians of western North America. Occasional Papers California Academy of Sciences 16:1-152.

Sredl, M.J., E.P. Collins, and J.M. Howland. 1997a. Mark-recapture of Arizona leopard frogs. Pages 1-20 *In:* M.J. Sredl (editor). Ranid frog conservation and management. Nongame and Endangered Wildlife Program Technical Report 121. Arizona Game and Fish Department, Phoenix, Arizona.

Sredl, M.J., J.M. Howland, J.E. Wallace, and L.S. Saylor. 1997b. Status and distribution of Arizona's native ranid frogs. Pages 37-89 *In:* M.J. Sredl (editor). Ranid frog conservation and management. Nongame and Endangered Wildlife Program Technical Report 121. Arizona Game and Fish Department, Phoenix, Arizona.

Sredl, M.J. 2005. *Rana yavapaiensis* (Platz 1984) lowland leopard frogs. *In:* Lanoo, M.J. (editor), Status and Conservation of U.S. Amphibians. Volume 2: Species Accounts. University of California Press, Berkeley, California.

Stebbins, R.C. 1951. Amphibians of western North America. University of California Press, Berkeley. 539 p.

Schwaibe, C.R., and P.C. Rosen. 1988. Preliminary report on the effect of bullfrogs in wetland herpetofaunas in southeastern Arizona. General Technical Report. USDA Forest Service. Pages 166-173.

Vitt, L.J., and D. Ohmart. 1978. Herptofauna of the lower Colorado River, USA: Davis Dam to the Mexican border. Proceedings of the Western Foundation of Vertebrate Zoology 2:35-72.