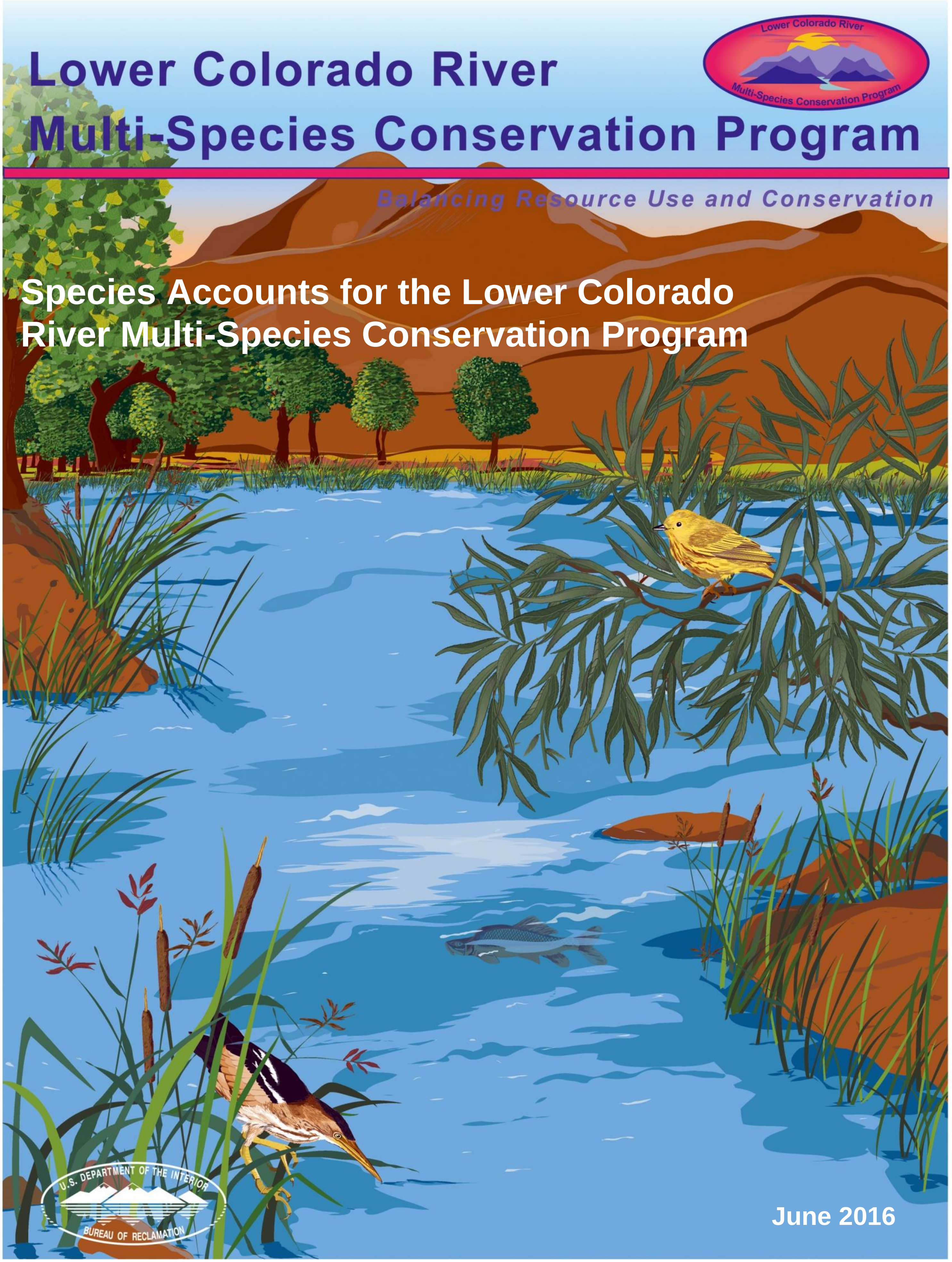

# Lower Colorado River Multi-Species Conservation Program

*Balancing Resource Use and Conservation*

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

U.S. DEPARTMENT OF THE INTERIOR
BUREAU OF RECLAMATION

June 2016

# 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 Local Governments Authority
Desert Wildlife Unlimited

### California Participant Group

California Department of Fish and Wildlife
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
Chemehuevi Indian Tribe

### Conservation Participant Group

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

# Lower Colorado River
# Multi-Species Conservation Program

## 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

June 2016

# ACRONYMS AND ABBREVIATIONS

| | |
|---|---|
| AGFD | Arizona Game and Fish Department |
| AOU | American Ornithologists' Union |
| BBS | Breeding Bird Survey |
| Bill Williams River NWR | Bill Williams River National Wildlife Refuge |
| BLCA | Beal Lake Conservation Area |
| BLM | Bureau of Land Management |
| CDFG | California Department of Fish and Game (now the California Department of Fish and Wildlife) |
| CDFW | California Department of Fish and Wildlife |
| Cibola NWR | Cibola National Wildlife Refuge |
| cm | centimeter(s) |
| CRIT | Colorado River Indian Tribe |
| CVCA | Cibola Valley Conservation Area |
| CW | cottonwood-willow |
| DNA | deoxyribonucleic acid |
| DWMA | Desert Wildlife Management Area |
| ft | foot/feet |
| ft/s | feet per second |
| FTHL ICC | Flat-tailed Horned Lizard Interagency Coordinating Committee |
| g | gram(s) |
| GBBO | Great Basin Bird Observatory |
| ha | hectare(s) |
| Havasu NWR | Havasu National Wildlife Refuge |
| HCP | Habitat Conservation Plan |
| Imperial NWR | Imperial National Wildlife Refuge |
| IUCN | International Union for Conservation of Nature |
| km | kilometer(s) |
| LCR | lower Colorado River |
| LCR MSCP | Lower Colorado River Multi-Species Conservation Program |
| LCRB | Lower Colorado River Basin |

| | |
|---|---|
| m | meter(s) |
| m/s | meters per second |
| mg/L | milligrams per liter |
| mm | millimeter(s) |
| mtDNA | mitochondrial deoxyribonucleic acid |
| NDOW | Nevada Department of Wildlife |
| NPS | National Park Service |
| NTU | nephelometric turbidity unit(s) |
| OHV | off-highway vehicle |
| OWSVRA | Ocotillo Wells State Vehicle Recreation Area |
| Reclamation | Bureau of Reclamation |
| RLFCT | Relict Leopard Frog Conservation Team |
| SVL | snout-to-vent length |
| TL | total length |
| UCRB | Upper Colorado River Basin |
| URTD | upper respiratory track disease |
| USFWS | U.S. Fish and Wildlife Service |
| WNS | white-nose syndrome |

## Symbols

| | |
|---|---|
| ° | degrees |
| °C | degrees Celsius |
| > | greater than |
| ≥ | greater than or equal to |
| < | less than |
| % | percent |
| ± | plus or minus |

# CONTENTS

# Introduction

The Lower Colorado River Multi-Species Conservation Program (LCR MSCP) was created to balance the use of Colorado River water resources with the conservation of native species and their habitats in compliance with the Endangered Species Act. This is a long-term (50-year) program to conserve at least 26 species along the lower Colorado River from Lake Mead to the Southerly International Boundary with Mexico through 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 its 50-year term. A Steering Committee, currently consisting of 57 entities, has been formed as described in the LCR MSCP Funding and Management Agreement to provide input and oversight functions in support of LCR MSCP implementation.

In 2008, Reclamation developed species accounts for 22 covered and 5 evaluation species listed in the HCP that use terrestrial, marsh, and riparian habitats and the 4 native fish species also covered under the LCR MSCP.[2] As new information is collected, it was envisioned that these species accounts would be periodically updated to reflect the current knowledge of these covered and evaluation species. This document serves as the first update of these species accounts and includes the humpback chub, which was not included in the first edition. It should also be noted that the information contained in the species accounts will be included in the conceptual ecological model (CEM) format in the future. The CEMs involve a more targeted literature review and assessment of scientific knowledge and create an explicit link between the science activities and restoration site management that will provide a framework for meeting the LCR MSCP's conservation measures. Species accounts for the southwestern willow flycatcher and yellow-billed cuckoo are not included in this update because separate CEMs were already in development for these species when this update began.

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, and behavior. These species accounts were developed to quantify existing knowledge for each species and identify

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[1] Bureau of Reclamation. 2004. Lower Colorado River Multi-Species Conservation Program, Volume II: Habitat Conservation Plan, Final. December 17. Available at: http://www.lcrmscp.gov

[2] Bureau of Reclamation. 2008. Species Accounts for the Lower Colorado River Multi-Species Conservation Program. Bureau of Reclamation, Lower Colorado Region, Boulder City, Nevada. Available at: http://www.lcrmscp.gov

information gaps that, if addressed, would better inform the creation and management of covered species' habitats, enabling the successful completion of conservation measures.

The species accounts will no longer be updated in the current format, but similar information will be incorporated into the CEMs as part of the CEM update process. This will be done on a periodic basis, 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 Final Science Strategy. [3]

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[3] Bureau of Reclamation. 2007. Lower Colorado River Multi-Species Conservation Program Final Science Strategy. Bureau of Reclamation, Lower Colorado Region, Boulder City, Nevada. Available at: http://www.lcrmscp.gov

# THREATENED AND ENDANGERED SPECIES

# Bonytail
# (*Gila elegans*)

## DISTRIBUTION

Bonytail (*Gila elegans*) 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 the species from the Upper Colorado River Basin (UCRB) was by Jordan (1891), with one specimen collected from the Green River. Subsequent historical accounts were limited largely to anecdotal and historical fishing creel interviews in conjunction with limited scientific collection information combined to demonstrate the once-expansive range of bonytail (U.S. Fish and Wildlife Service [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 [*Xyrauchen texanus*]” and “Flannelmouth Sucker [*Catostomus latipinnis*]”species profiles). Holden (1991) describes the effects of a large-scale rotenone treatment in the upper Green River, providing insight into 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 sporadic captures of very few individuals 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 Lower Colorado River Basin (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) document 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 broodstock – the progeny of which are presently stocked into Lakes Mohave and Havasu (Minckley et al. 1989, 1991; USFWS 2002) and a number of UCRB rivers. Very few wild bonytail had been captured prior to extirpation from the lower Colorado River; 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 main stem 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 non-native 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, see Tyus et al. (1982), Minckley and Deacon (1991), Mueller and Marsh (2002), and USFWS (2002).

# SYSTEMATICS AND MORPHOMETRICS

The following species description is based on information supplied by the 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 millimeters (mm), and may weigh over 1,100 grams. 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). Bonytail 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 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 the 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 chubs, and humpback chubs 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, 1998; Kaeding et al. 1990). 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 deoxyribonucleic acid (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 species tended to be found in higher-gradient, gravelly riverine sections, with some degree of habitat use similarities as described for flannelmouth suckers. For example, bonytail are widely characterized as being adapted to the swifter sections of the Colorado River, with an affinity for areas of high flow and rocky habitat. Available information suggests that adult bonytail use fast “flowing” water sections as well as eddies and pool habitats 1 to 3 meters (m) in depth (Bestgen et al. 2008; Vanicek 1967). Habitat selection of the species has been documented to coincide with habitats occupied by roundtail and humpback chubs (Vanicek 1967; Bestgen et al. 2008). Holden (1991), citing Flaming Gorge pre-impoundment surveys, notes that bonytail were apparently fairly common in the Flaming Gorge area of the upper Green River, a canyon-bound, relatively fast water section of the river. Valdez (1990) reports bonytail habitat use as being similar to that of the 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 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 used during the nighttime hours (Mueller et al. 2003; Mueller 2006; Marsh et al. 2013). Intensive telemetric surveillance suggests a high degree of site-specific habitat fidelity, with individually marked bonytail consistently returning each morning to the same zone, often to the exact cavity formed within the riprap-type shoreline (Marsh et al. 2013). These areas may simulate the boulder fields of many of the UCRB canyon areas where bonytail were once common.

A study conducted by Pimentel and Bulkley (1983) suggest that bonytail, when given the opportunity, tend to select water with high levels of total dissolved solids. They are able to persist in water with a total dissolved solids concentration of 4,700 milligrams per liter, the highest tolerance reported for any species of Colorado River *Gila*, suggesting an ability to persist despite anthropogenic water quality and habitat degradation. Telemetry studies conducted by Karam et al. (2012) show limited dispersal of bonytail stocked within the Bill Williams River National Wildlife Refuge compared to Cattail Cove State Park and Blankenship Bend that had 2.4 and 3.2 times greater dispersal, respectively. It is thought that water clarity may influence bonytail dispersal. Turbidity at the Bill Williams River National Wildlife Refuge ranged from 1.04 to 5.1 nephelometric turbidity units (NTU), Cattail Cove State Park averaged 1.0 NTU, and Blankenship Bend ranged from 0.48 to 0.50 NTU (Karam et al. 2012).

## 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). It is hypothesized that they use gravel-cobble habitats in lotic environments. Most recently in the LCRB, documentation of successful, natural reproduction in Cibola High Levee Pond suggests that the species selects 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 have displayed similar diel habitat shifts: adults used deeper habitats during the day and later form congregations along shoreline habitats (Mueller and Marsh 2002). Spawning occurs in March/April at Cibola High Levee Pond, in early May at Lake Mohave, and as late as early June in the upper Green River; the commonality of these dates appears to be temperatures ranging from 18–20 degrees Celsius (°C) (Mueller 2006).

### Larvae and Juveniles

Relatively little is known about the habitat needs of young bonytail. Similar to other native fishes, backwaters and other slackwater habitat types are thought to serve as important nursery areas for the young (USFWS 2002). Larval roundtail and humpback chubs 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. Relatively narrow nursery habitat requirements separate razorback suckers and Colorado pikeminnows from the non-endangered, more common species such as flannelmouth suckers and roundtail chubs. Therefore, it seems likely that in a riverine situation, bonytail may have a nursery habitat requirement that has not 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) document 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) classifies the species as being broadcast spawners and suggests 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) reports 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 main stem Colorado River.

Ripe bonytail have also been collected from lentic reservoir situations. As stated previously, Jonez and Sumner (1954) report active spawning of a large (approximately 500 individuals) aggregate of the species 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 the “Razorback Sucker [*Xyrauchen texanus*]” and “Flannelmouth Sucker [*Catostomus latipinnis*]” 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

The bonytail's diet comprises a wide variety of aquatic and terrestrial insects, worms, algae, plankton, and plant debris (Mueller and Marsh 2002; Marsh et al., 2013). This is corroborated by McDonald and Dotson (1960) and Vanicek (1967) who found Colorado River bonytail to feed omnivorously. More quantitative descriptions of the bonytail's 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 a gut analysis demonstrating that smaller size classes typically fed on zooplankton and invertebrates (Mueller et al. 2003). More detailed information is unavailable likely due to the overall rarity of this 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 the species' 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 grams. Ulmer (1983) used otoliths to determine that two Lake Mohave bonytail were 32 and 39 years old. Rinne (1976) estimates four Lake Mohave fish to be between 34 and 49 years old. Data suggest that bonytail are typically captured between 338 and 535 mm total length (USFWS 2002). In any case, the species is 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).

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# Desert Tortoise Mojave Population (*Gopherus Agassizii*)

## LEGAL STATUS

The Mojave population of desert tortoises (*Gopherus agassizii)* was listed as threatened on April 2, 1990. Critical habitat was designated on February 8, 1994, in portions of the Mojave and Colorado Deserts totaling 6.4 million acres (U.S. Fish and Wildlife Service [USFWS] 1994). A recovery plan for desert tortoises (Mojave population) was published in 1994 by the Desert Tortoise Recovery Team, headed by the USFWS (USFWS 1994). The USFWS published a revised recovery plan in 2011 (USFWS 2011a).

In June 2011, research was published documenting that the Mojave and Sonoran desert tortoise populations were two separate and distinct species. The originally recognized species, the Agassiz's (Mojave) desert tortoise, represents the listed populations naturally found west and north of the Colorado River in Utah, Nevada, northern Arizona, and California. The newly recognized species, the Morafka's (Sonoran) desert tortoise (*Gopherus morafkai*), represents populations naturally found east and south of the Colorado River, from Arizona extending into Mexico. The species covered under the Lower Colorado River Multi-Species Conservation Program (LCR MSCP) is the Mojave desert tortoise.

The 1994 recovery plan described a strategy for recovering desert tortoises, which included the identification of six recovery units, recommendations for a system of Desert Wildlife Management Areas (DWMAs) within the recovery units, and development and implementation of specific recovery actions. Maintaining high survivorship of adult desert tortoises was identified as the key factor in recovery, and because the list of threats to this species remains mostly unchanged since 1994, the requisite management or recovery actions were retained in 2011.

- *Recovery Objective 1 (Demography).* Maintain self-sustaining populations of desert tortoises within each recovery unit into the future.

- *Recovery Objective 2 (Distribution).* Maintain well-distributed populations of desert tortoises throughout each recovery unit.

- *Recovery Objective 3 (Habitat).* Ensure that habitat within each recovery unit is protected and managed to support long-term viability of desert tortoise populations.

The 2011 revised recovery plan builds upon the foundation laid by the 1994 recovery plan by emphasizing partnerships to direct and maintain focus on implementing recovery actions and a system to track implementation and effectiveness of recovery actions.  Strategic elements within a multi-faceted approach designed to improve the 1994 recovery plan are:

1. Develop, support, and build partnerships to facilitate recovery
2. Protect existing populations and habitat, instituting habitat restoration where necessary
3. Augment depleted populations in a strategic manner
4. Monitor progress toward recovery
5. Conduct applied research and modeling in support of recovery efforts within a strategic framework
6. Implement a formal adaptive management program (USFWS 2011a)

Desert tortoises are a species of special concern in Arizona, threatened in California, and a species of conservation priority in Nevada.

# DISTRIBUTION

## Historical Range

Desert tortoises were historically distributed in the Mojave 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

Desert tortoise populations have declined throughout their historical range and have been extirpated in parts of their range (Spang et al. 1988 *in* USFWS 1994; Berry 1978 *in* USFWS 1994).  Desert tortoises are divided into two species:  the Sonoran and Mojave, 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; Murphy et al. 2011).  The Mojave species occurs north and west of the Colorado River, and the Sonoran species occurs south and east of the Colorado River (USFWS 1994).

Mojave desert tortoises inhabit parts of the Mojave Desert in Inyo, Kern, Los Angeles, San Bernardino, and Riverside Counties in California; the 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). This species also inhabits the Colorado Desert, a division of the Sonoran Desert in Imperial, San Bernardino, and Riverside Counties, California (USFWS 1994). Their 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 (USFWS 1994). Six evolutionary distinct population segments were identified in the recovery plan: (1) Northern Colorado Desert, (2) Eastern Colorado Desert, (3) Upper Virgin River, (4) Eastern Mojave Desert, (5) Northeastern Mojave Desert, and (6) Western Mojave Desert. These population segments were based on genetics, morphology, behavior, ecology, and habitat use (USFWS 1994). In the 2011 revised recovery plan, the recovery units were revised, based on current genetic evidence and biomes, to the: (1) Upper Virgin River, (2) Northeastern Mojave, (3) Eastern Mojave, (4) Colorado Desert, and (5) Western Mojave (USFWS 2011a; 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).

A 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 occurred in the western part of the desert tortoise's range in the Mojave region (Berry 1984 *in* Tracy et al. 2004). In California's western Mojave Desert, 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), a decline resulted in an 88% reduced tortoise population; a similar 84% decline was reported for Johnson Valley (USFWS 1994 *in* NatureServe 2006). Furthermore, declines were reported in the Western Mojave Desert Recovery Unit (Tracy et al. 2004). A spatial analysis showed that areas of decline were greatest in portions of the Fremont-Kramer Desert and the northwestern part of the Superior Cronese DWMAs (Tracy et al. 2004).

In 2014, the USFWS used annual density estimates to compare a set of models that described desert tortoise abundance patterns among desert tortoise conservation areas (e.g., national parks, DWMAs, the Desert Tortoise Natural Area, etc.) and recovery units (USFWS 2014). The best model describing rangewide patterns in desert tortoise densities indicated different linear trends in different recovery units. In the original recovery plan for desert tortoises, the USFWS (USFWS 1994) expected that monitoring would help detect increasing population trends of no more than 2% per year over a 25-year period. They found much larger annual increases (greater than 19.7%) in the Northeastern Mojave Recovery Unit since 2004, with the rate of increase apparently resulting from increased survival of adults and subadults moving into the adult size class. The weight of evidence indicates that populations in the other four recovery units are declining: Upper Virgin River Desert (-5.1%), Eastern Mojave Desert (-5.8%), Western Mojave Desert (-9.8%), and Colorado Desert (-2.4%); however, populations in two desert tortoise conservation areas within this unit seem to be increasing (USFWS 2014).

Federal agencies that have jurisdiction over desert tortoise habitat in the Mojave region are the Bureau of Land Management (BLM), the National Park Service (NPS), the Department of Defense, the Bureau of Indian Affairs, and the Bureau of Reclamation (Reclamation). State parks and wildlife departments also manage desert tortoise habitat (USFWS 1994). The BLM is the primary land manager for desert tortoise habitat (USFWS 1994).

## Populations Within the LCR MSCP Planning Area

Mojave desert tortoises are present in Reaches 1–6 of the LCR MSCP planning area in an estimated 10,660 acres of desert scrub habitat (Reclamation 2004).

The 1994 recovery plan (USFWS 1994) described 14 DWMAs. Land ownership and tortoise densities were estimated for each DWMA:

1. The 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 land.

2. The 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. The land is owned by the BLM and military and private entities.

3. The 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. The land is owned by the BLM and military and private entities.

4. The Fenner DWMA is located in the Eastern Mojave Recovery Unit in San Bernardino County, California. Current densities of desert tortoises are 10 to 350 adults per square mile. Federal agencies own 67% of the land, State agencies own 5% of the land, and private entities own 28% of the land.

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

6. The Piute-Eldorado DWMA is located in the Eastern Mojave Recovery Unit in Clark County, Nevada. Current densities of desert tortoises are 40 to 90 adults per square mile. The land is owned by the NPS, BLM, and private entities.

7. The Beaver Dam Slope DWMA is located in the Northeastern Mojave Recovery Unit in Washington County, Utah, and Mohave County, Arizona. Current densities of desert tortoises are 5 to 56 adults per square mile. The land is owned by the BLM and private entities.

8. The Coyote Spring DWMA is located in the Northeastern Mojave Recovery Unit in Lincoln and Clark Counties, Nevada. Current densities of desert tortoises are 0 to 90 adults per square mile. The land is owned by the BLM, USFWS, and private entities.

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

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

11. The Fremont-Kramer DWMA is located in the Western Mojave Recovery Unit in Kern and San Bernardino Counties, California. Current densities of desert tortoises are 5 to 100 adults per square mile. The land is owned by the BLM, military and private entities, and State agencies.

12. The Ord-Rodman DWMA is located in the Western Mojave Recovery Unit in San Bernardino County, California. Current densities of desert tortoises are 5 to 150 adults per square mile. The Federal Government owns 65% of the land, and private entities own 35% of the land.

13. The Superior-Cronese DWMA is located in the Western Mojave Recovery Unit in San Bernardino County, California. Current densities of desert tortoises are 20 to 250 adults per square mile. The BLM owns 63% of the land, the Department of Defense owns 15% of the land, and private entities own 22% of the land.

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

# LIFE HISTORY

## General Description

A 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* Arizona Game and Fish Department [AGFD] 2002). It is also characterized by stocky limbs, forelimbs covered with large conical scales, and a short tail (AGFD 2002). The alveolar ridges of the upper jaw form a sharp angle with each other; jaw margins are serrated. The iris is greenish-yellow or yellow with brown near the outer edge, sometimes brown or mottled. The 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). Desert tortoises can be distinguished from the other three species in their 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). The carapace length of adults ranges from 20 to 36 centimeters (Stebbins 1985 *in* AGFD 2002). Desert tortoises reach their maximum size at 5–10 years of age (Murray and Klug 1996 *in* AGFD 2002).

A male can be distinguished from a female 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) a plastron that is more concave, especially in the femoral area, and (6) the dermal ossicles on the thigh and hindfoot are more developed (Auffenberg 1976 *in* Grover and DeFalco 1995; AGFD 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 millimeters (mm) in straight carapace length may be difficult by external morphology alone (NatureServe 2006).

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

Hatchlings are approximately 4.5 to 5.0 centimeters long and weigh approximately 20.0 to 27.0 grams. They are round in shape and are mustard yellow to brown in color. The edges of their scutes are typically brown, and the 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 the 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 of Mojave 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). A dominant male is characterized by a larger size, longer residency at a site, and past social interactions (NatureServe 2006). Male courting behaviors include 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). A female’s 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).

Desert tortoises lay their eggs from April through mid-July. They 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). The 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). The incubation period ranges from 85 to 125 days (Spotila et al. 1994 *in* NatureServe 2006). Temperatures at nest sites during this period need to be above 26 degrees Celsius (°C) and below 35 °C for the eggs to survive. Drier soils at nest sites are preferred. Soil moisture greater than 4% at nest sites 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 *in* AGFD 2002). Desert tortoise eggs are pale, elliptical to spherical, brittle shelled, and average 30–40 mm and 20–40 grams (NatureServe 2006). Eggs are laid in depressions 3–4 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

Desert tortoises feed 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). Desert tortoises will occasionally eat insects, which are a good source of lipids and protein (Grant 1936 *in* NatureServe 2006; Brown 1968 *in* NatureServe 2006; Okomoto 1995 *in* NatureServe 2006; H.W. Avery, personal communication *in* NatureServe 2006). Forage species selected by tortoises in the west Mojave Desert include: dwarf white milkvetch (*Astragalus didymocarpus*), widow's milkvetch (*Astragalus layneae*), desert suncup (*Camissonia boothii*), whitemargin sandmat (*Euphorbia albomarginata*), foothill deervetch (*Lotus humistratus*), and wishbone-bush (*Mirabilis bigelovii*) (Jennings *in* Boarman 2002a). In the east Mojave Desert, tortoises showed a preference for desert suncup, Panamint cryptantha (*Cryptantha angustifolia*), smooth desert dandelion (*Malacothrix glabrata*), prickly pear (*Opuntia basilaris*), New Mexico plumeseed (*Rafinesquia neomexicana*), common Mediterranean grass (*Schismus barbatus*), and wirelettuce (*Stephanomeria exigua*) (Avery 1998 *in* Boarman 2002a). An active adult individual requires approximately 21 kilograms of herbaceous forage per month (U.S. Department of the Interior 1991 *in* NatureServe 2006). Their diet is based on the presence and abundance of forage and consists primarily of annuals during spring and dry grasses and cacti during summer (Minnich 1972, 1979, 1982 *in* NatureServe 2006; Oftendal et al. 1995 *in* NatureServe 2006). Desert tortoises are 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 a 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

The activity period for desert tortoises varies by region, sex, and age class. The Mojave 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). They hibernate in burrows where they conserve water and energy (USFWS 1994) the remainder of the year. Some individuals may aestivate during dry periods in summer (NatureServe 2006). Between March through October, activity tends to be bimodal; morning activity begins around 0700 hours, with individuals retreating to their burrows from 1100 to 1600 hours (Berry 1975 *in* NatureServe 2006; Ruby et al. 1994 *in* NatureServe 2006). Desert tortoises maintain their body temperatures in the range of 25 to 35 °C (Zimmerman et al. 1994 *in* NatureServe 2006).

Desert tortoises demonstrate a delayed maturity and long life (MacArthur and Wilson 1967 *in* USFWS 1994). Existing data are consistent with the possibility that they 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). The home range size for desert tortoises varies from 5 to 50 hectares, 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). The home ranges of adults are usually larger than those of juveniles, and the home ranges of males are usually twice the size of that of females (O’Connor et al. 1994 *in* NatureServe 2006). The 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 lives 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 grams. The size of the scat indicates the size of the tortoise. Scat may serve as a territorial marker 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 (USFWS 1994). The average annual growth of healthy, non-threatened desert tortoise populations ranges 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 the Mojave desert tortoise 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).

A 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*), Mojave 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

Mojave desert tortoises are found primarily in Mojave 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*) woodlands 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 meters (Luckenbach 1982 *in* AGFD 2002; Collins et al. 1983 *in* AGFD 2002). The basic habitat requirements of desert tortoises 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 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 spring and summer, when tortoises are active, have the following characteristics: (1) usually larger and longer than the tortoise, often extending 1–8 feet in length, (2) mean floor declinations of 15 degrees, (3) opening faces north, northwest, or northeast, (4) often under a shrub, and (5) have a single opening (Burge 1978 *in* NatureServe 2006; Woodbury and Hardy 1948 *in* NatureServe 2006). Burrows used in winter, when desert tortoises hibernate, have the following characteristics: (1) extend 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 can be used to supplement existing populations and to create new populations in the desert tortoise's historic range (Tracy et al. 2004). The method can also be used when relocating tortoises out of harm's way off a project site. Recent studies on Mojave desert tortoise translocation have shown promise. However, these studies have occurred over durations of less than 5 years, various risks have not been fully evaluated, and long-term success has not been demonstrated (USFWS 2011b). Ongoing research indicates that translocation methods, including selection of release sites, influence desert tortoise movements following release (Germano 2011), and ensuring only healthy animals are used is also critical (USFWS 2013). The USFWS has developed guidance for translocations (USFWS 2011a) and health assessments (USFWS 2013).

## Survey Methods

Desert tortoises are sparsely distributed, and a certain number are underground and not visible at any time. When they are out of their shelters, they are cryptically colored and shaped. Their behavior also does not draw attention to them. Sampling and survey methods have developed over time to improve detectability and estimates.

### Determining Presence/Absence and Estimating Abundance Within a Project – Pre-Project Field Survey

In 2009, the USFWS updated survey protocols for determining the presence/ absence and abundance of desert tortoises for projects occurring within this species' range on Federal and non-Federal lands and to provide a standard method for reporting survey results. Information gathered from these procedures will help: (1) determine the appropriate level of consultation with the USFWS and the appropriate State wildlife agency, (2) determine the incidental take of desert tortoises resulting from proposed projects as defined by the Endangered Species Act and the California Endangered Species Act, and (3) minimize and avoid take. This involves a calculation of: (1) the size of the project area, (2) the probability that a tortoise is above ground, and therefore observable, and (3) the number of adult tortoises (> 160 mm midline carapace length) that were observed in the project area (USFWS 2009).

### Population Trend Surveys – Line Distance Sampling

Before tortoises were listed as threatened under the Endangered Species Act, populations were monitored either using strip transects (Luckenbach 1982), in which indications of tortoise presence (live or dead tortoises, scat, burrows, or tracks) were converted to tortoise abundance categories based on calibration transects conducted in areas of better-known tortoise density, or by using capture-recapture population estimates on a limited number of (usually) 1-square-mile study plots (Berry and Nicholson 1984). Although data have continued to be collected on transects and study plots in recent years, these methods suffer statistical deficiencies and/or logistical constraints that render them unsuitable for monitoring trends in abundance applicable to entire recovery units (Tracy et al. 2004). In 1999, the Desert Tortoise Management Oversight Group endorsed the use of line distance sampling (Buckland et al. 2001) for estimating rangewide desert tortoise density.

Distance sampling methods correct population estimates for the proportion of the population that was hidden and not visible and for the proportion that was not detected although tortoises were on the surface. The line transect method, a modification of the strip transect method in which an observer travels down the centerline of a strip of defined length ($L$) and width ($2w$, where $w$ equals the distance from the center to the edge of the strip) and records every tortoise observed ($n$), is used to monitor desert tortoises. Density ($D$) is then simply $n$ divided by the area searched ($2wL$). With this method, it is assumed that all tortoises within the strip are located (USFWS 2012). From 2001 to 2005, and again from 2007 through 2012, desert tortoise populations in four of the five recovery units have been part of a coordinated, rangewide monitoring program using line distance sampling. (The Upper Virgin River Recovery Unit is monitored by the Utah Division of Wildlife Resources.)

## Threats

Desert tortoises have been extirpated or have severely declined from the western and northern parts of their geographic range in California (Antelope, Indian Wells, and Searles Valleys) (Jacobson 1994 *in* USFWS 1994). Desert tortoises are subject to multiple threats simultaneously in many parts of their 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 desert tortoises are habitat destruction, degradation, and fragmentation from urban and agricultural development; livestock grazing; mining; invasion of non-native plants; fire; and off-highway vehicle (OHV) use (Jacobson 1994 *in* USFWS 1994). Direct mortality or injury of desert tortoises caused by humans and disease are other major threats to this species (Tracy et al. 2004).

Agricultural development causes widespread reduction of the water table, increases raven populations, clears native vegetation, introduces pesticides and fertilizers to habitat, and provides a seed source for non-native plants (USFWS 1994). For example, Russian thistle (*Salsola tragus*) 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 California Department of Fish and Game 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 the rate of non-native species invasion (Jacobson 1994 *in* USFWS 1994). The reduction of native perennial grasses reduces forage and protein availability for 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 rayless goldenhead (*Acamptopappus sphaerocephalus*), water jacket (*Lycium andersoni*), spiny hopsage (*Grayia spinosa*), winterfat (*Ceratoides lanta*), and Mojave woodyaster (*Machaeranthera tortifolia*) (Bentley 1898 *in* USFWS 1994; Frenkel 1970 *in* USFWS 1994; Humphrey 1958, 1987 *in* USFWS 1994; Humphrey 1987 *in* USFWS 1994; Rowlands, unpublished BLM 1980 *in* USFWS 1994; USFWS 1994). Livestock grazing has contributed to the spread of non-native plants such as redstem stork's bill (*Erodium cicatarium*), common Meditarranean grass, Arabian schismus (*Schismus arabicus*), brome (*Bromus* sp.), and prickly Russian thistle (*Salsola iberica*) (Kay et al. 1988 *in* USFWS 1994). Fire is also a threat to desert tortoises. Non-native, ephemeral plants have invaded the Mojave 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, non-native 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 tortoise's active season. Previously rare, the frequency of spring fires is now on the increase due to the encroachment of non-native 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 desert tortoises (USFWS 1994). Their populations are depleted up to a mile or more on either side of roads when the average daily traffic is greater than 180 vehicles (Nicholson 1978 *in* USFWS 1994). Dirt roads, which do not get much vehicle use, can cause a 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).

OHVs in the desert tortoise's 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 an increase in fires (USFWS 1994). Recent research in the Mojave Desert has demonstrated that the 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). Construction, operation, and maintenance of these facilities include 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, personal communication *in* USFWS 1994). Utility and natural gas lines disturb desert tortoise habitats in areas that are 50 to 125 feet wide surrounding the point where the lines are installed (USFWS 1994).

Military activities in the desert tortoise's historical range also 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, and shell casings); and chemical distribution.

Diseases, such as upper respiratory track disease (URTD) and shell disease, are threats to desert tortoises (Jacobson 1994 *in* USFWS 1994; Berry 1990 *in* USFWS 1990; Avery and Berry *in* USFWS 1990). The available evidence indicates that URTD is probably the most threatening infectious disease for desert tortoises, though a number of diseases are found in the population (USFWS 2013). At least two pathogenic species of *Mycoplasma* known to cause URTD in desert and gopher tortoises have been identified (*Mycoplasma agassizii* and *Mycoplasma testudineum*) (Brown et al. 1995, 1999, 2001, 2002). URTD was documented as a major cause of tortoise mortality in the Mojave desert tortoise, particularly in the western Mojave (Berry 1990 *in* USFWS 1994). From 1979 to 1992, the total population density in the Desert Tortoise Research Natural Area in the western Mojave Desert decreased by 76%. From 1988 to 1992, this decline was clearly contributable to URTD (Berry 1997). It is probable that pathogenic and non-pathogenic 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 their 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* USFWS 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 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 shell disease, cutaneous dyskeratosis, 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 a decline of desert tortoises (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 of captive tortoises in the wild poses a threat to native populations from genetic pollution, the potential for introducing or spreading disease, and disturbance to the social structure of the host population (USFWS 1994). The outbreak of URTD in the Mojave desert tortoise 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 Texas tortoises (*Gopherus berlandieri*), or another population unit, such as introducing a Sonoran tortoise in the Mojave population, also poses threats to desert tortoises (USFWS 1994).

Illegal dumping is another threat to desert tortoises (USFWS 1994). Desert tortoises have 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, personal communication, *in* USFWS 1994; K. Dahl, personal communication *in* USFWS 1994). Objects can be lodged in the gastrointestinal tract, causing death (USFWS 1994). Balloons that are released in mass are also threats to desert tortoises (USFWS 1994).

Predators of desert tortoises 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; Farrell 1989 *in* USFWS 1994). Feral and domestic dogs and cats are also predators of desert tortoises (Causey and Cude 1978 *in* USFWS 1994; Berry 1979 *in* USFWS 1994). The common raven, whose numbers have increased in the Mojave and Colorado Deserts since 1968, is a major predator of juvenile tortoises (Jacobson 1994 *in* USFWS 1994). Raven populations have increased by nearly 800–1,400% in the Mojave and Sonoran Deserts over the past 37 years (Boarman and Kristan 2006a). Raven population growth rates, dispersal rates, and local abundance continue to be the highest in the west Mojave Desert (Boarman and Kristan 2006a). Berry (1990 *in* USWFS 1994) believes that the increase of common raven populations has effected juvenile recruitment. Adult tortoises 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 desert tortoises 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).

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# Humpback Chub (*Gila cypha*)

## GENERAL

Humpback chubs (*Gila cypha*) are medium-sized (30 to 41 centimeters [cm] standard length as adults) freshwater minnows of the family Cyprinidae. These fish are described as both “bizarre” and “streamlined” because of their unusual shape; however, these physical adaptations also make these fish extremely well suited for life in the high, turbulent flows characteristic of the historical main stem of the Colorado River and its canyon-bound tributaries (Minckley 1973; U.S. Fish and Wildlife Service [USFWS] 1987; Minckley and Marsh 2009). Their bodies lack scales (except along the lateral line), which minimize friction in strong currents (Miller 1946). It is generally believed that these adaptations aid the animal in negotiating turbulent water.

Colors of humpback chubs vary depending on the environment. The species was originally described as being brownish to pinkish brown on the sides and belly and yellowish brown along the back (Miller 1946). More recent accounts describe humpback chubs as being silvery gray overall (when taken from turbid water), while clear water inhabitants may range from dark, olive gray to brownish, transitioning to silver laterally, with white undersides (Minckley and Marsh 2009).

The historical range of humpback chubs includes the Green and Colorado Rivers and their major tributaries in Wyoming, Utah, and Colorado. These tributaries include the Little Colorado, Yampa, and Green Rivers. Just as current-day humpback chub populations are mainly restricted to canyon-bound, whitewater reaches, it is believed historically that the fish was mainly confined to the same areas. The downstream (southerly) extent of their historical range is the Colorado River at current-day Lake Mohave but is only supported by a single archaeological record. Most observations of humpback chubs have been recorded from the unregulated Little Colorado River and Colorado River main stem in the Grand Canyon and in the unregulated Yampa and Green Rivers (Minckley and Marsh 2009; Miller 1946; Kaeding and Zimmerman, 1983). Based on the rarity of humpback chubs in fisheries collections following their initial discovery in 1946, it is unlikely that this species was ever abundant and widespread during the 20th century (Holden and Stalnaker 1975).

## Diet

Food habits of humpback chubs have not been thoroughly studied (Minckley and Marsh 2009); however, food items known to be consumed by the species include a variety of benthic aquatic invertebrates, including the families Chirinomidae and Simuliidae, as well as small fish (fathead minnow [*Pimephales promelas*]) (Kaeding and Zimmerman 1983). Larval humpback chubs in the Little Colorado River have been reported to feed on terrestrial insects, algae, and undifferentiated organic debris (Childs et al. 1998 *in* Minckley and Marsh 2009).

## Age and Growth

Humpback chubs grow rapidly during early life and reach maturity in 2–3 years to a maximum of about 51 cm (Mueller and Marsh 2002). Fish taken from the Little Colorado River were estimated to reach approximately 100 millimeters (mm) at age-1 fish and 250–300 mm by age-3. Similar size ranges were observed for humpback chubs taken from the Little Colorado River as well as its confluence with the Colorado River (Kaeding and Zimmerman 1983).

## Reproduction

Humpback chub females reach reproductive maturity at lengths between 250 and 300 mm (Kaeding and Zimmerman 1983), which generally corresponds to 3 years of age. Male reproductive maturity is believed to occur at the same approximate size range, although sexually ripe males have been observed as small as 205 mm total length. Gonad development occurs from December through April. Spawning is believed to occur between April and May on the Little Colorado River and between April and June on the Yampa River (Kaeding and Zimmerman 1983; Modde and Smith 1995). Sexually ripe males are found on the Little Colorado River between February and May. The timing of spawning on the Yampa River is believed to be controlled more by the hydrograph than by a specific time of year (Modde and Smith 1995).

Humpback chub eggs require incubation temperatures above 10 degrees Celsius (°C), but below 30 °C to hatch, with laboratory experiments reporting optimal hatching success when eggs are incubated at 20 °C (Marsh 1985). Water temperatures of the Colorado River at the Grand Canyon average from 2 to 18 °C, which is believed to limit or preclude successful spawning outside of the Little Colorado River (Converse et al. 1998; Paukert et al. 2006).

## Hybridization

The genus, *Gila,* is a taxonomically complex group of Colorado River native fishes. Complexity arises from the strong occurrence of hybridization among congeners. This hybridization is especially common between humpback chubs and bonytail (*Gila elegans*) (Gerber 2001; Holden and Stalnaker 1970). Hybridization results in a broad range of subtle morphological variability and a high level of uncertainty in early field identification (Minckley 1973; USFWS 1987; Holden and Stalnaker 1970). As recently as 1990, taxonomic techniques to positively distinguish between *Gila* species and their hybrids in the Upper Colorado River Basin were acknowledged by researchers as lacking (Karp and Tyus 1990.), and in at least one case in the 1980s, field collections were reported as simply *Gila* spp. (Haynes et al. 1985).

# SPECIES THREATS

A combination of factors has been blamed for the decline of humpback chubs, including habitat modification, competition with and predation by introduced fish species, pollution and eutrophication, parasitism, changes in food base, and fishing pressure. Hybridization with congeneric chubs has also been suggested as an adverse factor.

## Habitat Modification

During the early 1900s, water development projects drastically modified the habitat of the main stem of the Colorado River. Construction of large dams transformed sediment-laden, swift-flowing sections of the main stem of the river, altering its flow regime (Mueller and Marsh 2002). These changes removed most of the sediment from the river and altered temperatures downstream from the dams (such as Glen Canyon Dam, which impounds Lake Powell). The combined effects of the altered hydrology created an environment that is clearer and colder, which does not support successful spawning for humpback chubs or most native fish species (Minckley and Marsh 2009).

In the last 21 kilometers of the Little Colorado River, immediately upstream of the Colorado River confluence, humpback chubs have been numerically dominant for some three decades (Minckley and Marsh 2009). These authors attribute this domination to the unaltered condition of the river channel, which likely resembles historical conditions (Kaeding 1983). A remnant population of humpback chubs has also been studied in the Yampa River at Dinosaur National Monument, a location with similarly natural hydrology. Researchers also attribute the persistence of the species at that location to relatively unaltered hydrology, although altered temperatures may still limit spawning success there (Karp and Tyus 1990).

## Hybridization

The ability of *Gila* to hybridize is believed to have been beneficial, historically, in adapting to changing environmental conditions found in the Southwest by enabling local populations to quickly adapt to environmental fluctuations (Gerber 2001). The question has been considered whether hybridization post-dam construction in large reservoirs may be responsible for increased hybridization due to increased connections between previously isolated populations where large reservoirs, such as Lake Powell and Flaming Gorge Reservoir, now cover historically main stem and tributary river habitats (Holden and Stalnaker 1970).

## Non-Native Species

Following construction of two Upper Colorado River Basin dams, State fish and wildlife agencies conducted rotenone (fish toxin) applications in the upper Green River and its tributaries to eradicate native fishes and develop a sport fishery in the new Flaming Gorge Reservoir (Vanicek and Kramer 1969; Quartarone 1995).

Predation by, and competition with introduced, non-native fishes, is presently acknowledged as a primary threat to humpback chubs, particularly the introduction of rainbow trout (*Oncorhynchus mykiss*) and brown trout (*Salmo trutta*) (Runge 2011; Marsh 1997) and are prioritized by some authors as the most critical risk to the survival of native fishes in the Southwestern United States in general (Minckley and Marsh 2009).

High levels of habitat overlap between humpback chubs and highly abundant channel catfish (*Ictalurus punctatus*) have been observed, with researchers pointing to marks on humpback chubs resembling catfish bite marks, which supports their conclusion that channel catfish on the Yampa River may represent a significant risk to humpback chub populations (Karp and Tyus 1990.). Humpback chub remains have been found in the stomachs of rainbow trout, channel catfish, and black bullhead (*Ameiurus melas*) (Marsh 1997).

# CONSERVATION OUTLOOK

Humpback chubs were a member of the original endangered species list prepared by the Office of Endangered Species in 1964 (USFWS 1987). They were listed under the Endangered Species Act in 1974 (Federal Register 1974) and designated as endangered in Colorado and as a protected species in Utah (USFWS 1987).

Under the Upper Colorado River Basin Recovery Implementation Plan, goals for recovery and delisting of three native fish species were established: razorback suckers (*Xyrauchen texanus*), bonytail, and humpback chubs, with an estimated timeframe of 15 years from the publication's date. The major elements of the plan comprised actions to manage and improve habitats, augment native fish species through stocking and managing non-native species, and to conduct research (USFWS 1987).

In 1988, the Upper Colorado River Endangered Fish Recovery Program was established in order to work toward recovery of humpback chubs and three other main stem Colorado River native fishes (bonytail, Colorado pikeminnow [*Ptychocheilus Lucius*], and razorback suckers) (Upper Colorado River Endangered Fish Recovery Program 2007). The major elements of the program include instream flow identification and protection, habitat restoration, non-native fish management, propagation and stocking, research and monitoring, information and education, and program management.

Downlisting humpback chubs from endangered to threatened requires maintaining the six existing populations with "no net loss," with one core upper basin population greater than 2,100 adults and one core lower basin population of greater than 2,100 adults, over a 5-year monitoring period. Delisting the species would require maintaining these standards for an additional 3 years beyond downlisting. Recovery goals were approved in 2002 and are updated every 5 years (Upper Colorado River Endangered Fish Recovery Program Web site, n.d.).

Removal and suppression of non-native fishes has been central to research and management in the Upper Colorado River Basin. Experimental removal of non-native fishes in Grand Canyon was undertaken from 2003 to 2006 and subsequently linked to increased recruitment of humpback chubs (U.S. Geological Survey 2011). In 2011, a multi-party decisionmaking effort was completed to develop management scenarios to address non-native fish control in the context of scientific uncertainty and conflicting value systems (Runge et al. 2011).

Management of releases to benefit endangered fishes is an additional major component of conservation in the Upper Colorado River Basin. Flaming Gorge Dam was modified in 1978 to discharge water from higher, warmer reservoir levels, resulting in warmer tailwater releases, although resulting benefits to humpback chubs are not readily identified in the literature (Holden 1980). Extensive flow management studies have been undertaken to determine best practices for managing flows on the Yampa, Green, and Colorado Rivers to benefit humpback chubs and other endangered native fishes (Modde and Smith 1995; Upper Colorado River Endangered Fish Recovery Program 2007).

Since 2009, the USFWS has been translocating juvenile humpback chubs from the Little Colorado River to other Grand Canyon tributaries, namely Shinumo and Havasu Creeks. The effort is intended to promote the survival of the species in Grand Canyon by establishing additional populations (National Park Service, n.d.).

Under the Lower Colorado River Multi-Species Conservation Program, $500,000 is being contributed over its 50-year cycle to support existing humpback chub conservation programs (Bureau of Reclamation 2004).

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# Razorback Sucker (*Xyrauchen texanus*)

## DISTRIBUTION

Razorback suckers (*Xyrauchen texanus*) 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) note particularly high razorback sucker abundance in the Lower Colorado River Basin (LCRB) near Yuma, Arizona; however, Bestgen (1990) indicates that razorback suckers 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 suckers 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 suckers are typically collected in sand-bottomed, low-gradient, flatwater reaches outside of their spawning period. Razorback suckers 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). Catches of razorback suckers were reported often from the early 1940s through the early 1980s in the LCRB (Minckley 1983; Marsh and Minckley 1989). Today, the Lake Mohave population is largely supported by stocking captive-reared fish (Marsh et al. 2003, 2005). More recently, over 12 million razorback suckers 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, 2004; Albrecht et al. 2010a).

# HISTORICAL HABITAT MODIFICATIONS

Numerous researchers have identified that the major factor contributing to the decline of razorback suckers and other large-river fishes has been the construction of main stem 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; Muth et al. 2000). This change in the physical environment presumably allowed for an increase in competition and predation from non-native fishes, which are successfully established in the Colorado River and its reservoirs and have also contributed to recruitment failure and population declines of native fishes (Minckley et al. 1991, 2003; Modde and Haines 2005; Mueller 2006; Carpenter and Mueller 2008). 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), and Tyus and Karp (1989, 1990).

# SYSTEMATICS AND MORPHOMETRICS

*Xyrauchen* is one of three monotypic genera of the family Catostomidae. According to Bestgen (1990) and the USFWS (1998), Abbott (1861) originally describes razorback suckers 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), and 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 14–15, primary anal fin rays are 7, vertebrae are 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 kilograms, but are more typically found within the 400—700 mm

> TL range, weighing less than 3 kilograms. During spawning, razorback sucker 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) describes razorback sucker 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 suckers and other native Colorado River catostomid species has historically been documented to occur. Most often, razorback suckers have been shown to hybridize with flannelmouth suckers, 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) uses allozymic data to directly quantify presumed introgression in the range of 0–5% toward flannelmouth suckers and 0–3% toward razorback suckers. Furthermore, in a natural river setting, Ryden (2000) noted adult flannelmouth suckers were captured consistently over the same cobble-bottomed riffles as mature, adult razorback suckers, suggesting concern for possible hybridization in San Juan River populations due to an overlap in physical habitat usage during the adult life stage of both species. Hybridization of razorback with flannelmouth suckers is also suspected in Lake Mead at the Colorado River inflow (Albrecht et al. 2010b, Kegerries and Albrecht 2011, 2013).

## Habitat

### Adults

Historically, razorback suckers 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 suckers (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 suckers 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 suckers changes seasonally. Tyus and Karp (1990) document habitat use by adult razorback suckers to consist of flooded areas during spring months. Radio telemetry efforts by Tyus (1987) identifies adult fish using near-shore runs during spring, but they subsequently shifted habitat use during summer to shallow waters associated with submerged midchannel sandbars, with little use of backwaters. This suggests that the use of backwaters by razorback suckers may be overstated and an artifact of relatively easy capture with electrofishing rather than actual habitat use and preference. Osmundson and Kaeding (1989) report adult razorback suckers 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 Upper Colorado River Basin (UCRB) spawning period.

More detailed information on razorback sucker habitat use, needs, and selection is provided by Ryden (2000), based on radio-telemetered razorback suckers 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 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 degrees Celsius [°C], main channel = 9.8 °C). The 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 suckers 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 shoals or backwaters were the most commonly selected habitat types. Furthermore, in April, mean water depth at fish locations was 2.3 ft, with razorback suckers seeking warmer temperatures (13.0 °C) than the main channel (12 °C). The mean bottom velocity was found to be 0.6 ft/s, and the average column velocity was 1.0 ft/s. During May, habitat selection demonstrated that razorback suckers showed a strong preference for eddy habitats located along the inside of large river bends. Also during May, razorback suckers displayed a strong affinity for midchannel 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 the "Spawning" section below). Mean water depth usage in May was 3.3 ft, and temperatures in habitats used by razorback suckers 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 the use of inundated vegetation. During high-flow periods, radio-telemetered razorback suckers were found using 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 used in June averaged 3.9 ft, and June was the last of three consecutive months in which water temperatures at fish locations were warmer than adjacent main channel areas (15.0 versus 14.8 °C). The 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 used. Ryden (2000) reports the mean bottom velocity during July to be 0.7 ft/s, the mean column velocity to be 1.6 ft/s, and the average temperature occupied by razorback suckers to be 21.1 °C.

During the post-runoff summer and fall months (August through October) Ryden (2000) found razorback suckers 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 during the summer-fall base flow period. For example, during August, razorback suckers were typically found using main channel runs and shoal runs. Likewise, depths used by razorback suckers 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). The mean water depth at fish locations was 3.8 ft, and the mean temperature at fish locations was reported as 5.3 °C. The 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 during early December when daytime water temperatures surpassed 3.0 °C. However, later in December, as temperatures began to decline, radio-tagged razorback suckers were observed using edge pools only, and the 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 suckers were only found using 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 suckers once again became fairly active and selected edge pools, main channel runs, eddies, and shore runs. The water depth at fish locations averaged 3.7 ft. The mean velocity at point of contact was 1.0 ft/s on the bottom as well as at midcolumn. 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.

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 suckers is also typified by seasonal shifts in preferences. Tyus (1987) notes that during summer, razorback suckers typically were found using waters with velocities averaging 0.5 meter per second (m/s), while in the winter months, adult fish were typically found in currents moving at 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 suckers use 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 suckers in a natural river setting do not appear to use solely backwater habitat types, although it appears that these habitats are important during specific times of the year. Lastly, adult razorback suckers have been reported to select waters with shallower depths during the summer months (0.9–1.65 meters [m]) while typically using deeper depths during the winter months (1.65–2.16 m) (Osmundson and Kaeding 1989).

In contrast, hatchery-raised, sonic-tagged razorback suckers 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. Wydoski and Lantow (2013) report observations similar to previous authors but found that fish captured within 4 years of stocking were found in backwaters, while fish that were at large for 5 or more years were found in the main channel. They additionally found that fish size may also play a role in habitat selection, observing that razorback suckers less than 400 mm were more likely to be contacted in backwater habitats and that razorback suckers larger than 450 mm were more likely to be contacted in the main channel. More specific to findings presented in Bradford et al. (1998) and Bradford and Gurtin (2000), Slaughter et al. (2002) reports that adult razorback suckers prefer large, irregularly shaped backwaters with a mean depth greater than 1.5 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 suckers 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 two basins. 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 suckers 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 suckers, 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. Mueller (2006) supports this statement in part, emphasizing the importance of large off-channel backwaters as a refuge for native fishes where water temperatures are generally warmer than the main channel and where a greater number of food organisms such as crustaceans, invertebrates, and zooplankton are supported.

One of the current habitat types presently occupied by populations of razorback suckers in the LCRB are the lentic areas imposed by various impoundments. Although these fish historically occupied main channel riverine and flood plain habitats, their ability to survive in artificial reservoirs is likely due to an evolutionary predisposition to lake environments (Mueller and Marsh 2002). 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 suckers use a wide variety of habitats, including vegetated areas, pelagic and littoral shoreline habitats, and substrates ranging from silt and sand to gravel and cobble (Albrecht et al. 2008a). Adult razorback suckers 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 the winter months (less than 30 m from shore during peak spawning activity). However, during the 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 and Holden 2003, 2004). Thermal preferences for adult razorback suckers were 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 suckers was 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 suckers, as those in early life stages have been reported using backwater habitats with dissolved oxygen levels approaching 2.0 mg/L at times, with critical dissolved oxygen levels dependent upon water temperatures (Modde 1996; Modde et al. 2001). Additionally, Stolberg (2012) found very little difference between growth rates for larval razorback suckers exposed to dissolved oxygen levels ranging from 3.0 to 7.8 mg/L. Growth at this early life stage may be less affected by environmental conditions when food sources are abundant.

## Spawning

Razorback suckers are broadcast spawners that release and fertilize their eggs near the bottom of the water column so that incubation can occur in the protected interstitial spaces of the substrate. The spawning season for razorback suckers has been reported to begin as early as November in some LCRB reservoirs (Bozek et al. 1991; Kretschmann and Leslie 2006) and to continue into June in some populations of the UCRB (Bestgen et al. 2002; Bestgen and Haines 2010). In upper basin riverine habitats, ripe razorback suckers have been collected from mid-April to mid-June, typically over a very limited timeframe (4–6 weeks) (Tyus 1987; Osmundson and Kaeding 1989; Tyus and Karp 1989, 1990; Bestgen 1990; Muth et al. 1998a). Back calculation of spawning and hatching dates from larval razorback suckers captured in the UCRB from 2002–07 also document spawning over a relatively short period (mid-April through May) (Osmundson and Seal 2009). However, in lentic lower basin habitats such as Lakes Mead and Mohave, the majority of spawning is generally carried out over cobble shoals from January through April (Mueller and Marsh 1998; Kegerries et al. 2009; Kesner et al. 2012; Albrecht et al. 2013) when water temperatures are typically within the range of 10–15 °C (Bestgen 1990). Male razorback suckers 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). Spawning has been documented in a wide range of water temperatures (6–21 °C), generally in shallow (< 1.0 m), low-velocity (< 1.0 m/s) habitat (Valdez et al. 2012). Spawning razorback suckers have also been collected over a variety of substrates, although the majority of spawning individuals tend to be captured over clean gravel and cobble-sized or rocky substrates (Douglas 1952; Tyus 1987; Bozek et al. 1990; Tyus and Karp 1990; Minckley et al. 1991; Kegerries et al. 2009). In UCRB rivers, spawning occurs during the ascending limb of the hydrograph (Modde 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 suckers 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 suckers have also been documented to use warmer backwater habitat types post-spawn apparently to recover and feed (Modde and Irving 1998).

Reservoir-spawning razorback suckers 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 and Holden 2003, 2004), Lake Mohave (Bozek et al. 1984, 1990; Marsh and Langhorst 1988; Mueller 1989, Lake Havasu (Douglas 1952; Minckley 1983), Senator Wash Reservoir (Medel-Ulmer 1980), and likely other locations. Razorback sucker spawning has also been observed in several LCRB backwaters and artificial ponds (Pacey and Marsh 1998). 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 m in Lake Mead but typically occurs in less than 2 m of water (Minckley et al. 1991; Holden et al. 1997, 1999).

Spawning fishes 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 fishes presently occurs only in a few sites with abundant nearby vegetation and turbidity that serves as cover for larvae. Razorback sucker spawning locations within Lake Mead may shift during the year, or between years, due to changes in reservoir elevation (Kegerries et al. 2009), but the locations generally remain near the turbid river inflow areas. 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 and Holden 2003, 2004). This suggests that although razorback suckers 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 suckers in spawning condition from gravel bars in water typically 1 m deep. They report substrate used by spawning razorback suckers 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 centimeters, deeper depth to embeddedness, and fewer fine materials than the control riffle. This suggests that razorback suckers 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 radio telemetry data to document the spawning activity of individually tagged fish at different spawning locations during their 1993–95 study, suggesting that razorback suckers in the Green River represent a single reproductive population. Tyus and Karp (1990) used radio telemetry to document the importance of flooded lowlands and other slackwater habitats as resting/feeding areas for razorback suckers during their 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 suckers 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 and Holden 2003, 2004; Modde et al. 2005).

In the LCRB, Mueller (1989) provides insight into 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 suckers 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 suckers were visually observed upstream of 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.

## 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, and 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 and Holden 2003, 2004).

In reservoirs and rivers, larvae razorback suckers have been observed using the interstitial spaces of the existing substrate for cover during daylight hours and emerging to feed at night (Bestgen 1990; Bozek et al. 1990). In the Green River, larval razorback suckers apparently have an affinity for backwater and flooded bottomland habitats (Tyus 1987; Muth et al. 1998b). Historically, high spring flows flooded low-lying areas along the river and redistributed recently emerged and drifting larval razorback suckers into these food-rich backwaters and other seasonally flooded bottomlands, providing unique nursery habitats for razorback suckers (Tyus and Karp 1989, 1990; Modde 1996; Modde et al. 1996, 2005; Wydoski and Wick 1998). Hedrick et al. (2009) found that entrainment of larval razorback suckers in the flood plain wetlands of the middle Green River occurred frequently and that is was most effective when water was able to enter sites on both the ascending and descending limbs of the hydrograph. Laboratory experimentation has documented the importance of backwater habitats for larval razorback suckers by evaluating nocturnal drift tendencies of young razorback suckers 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 main stem 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 the 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 suckers 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 (Brandenburg and Farrington 2010). This is consistent with the observations of other researchers who identified quiet shoreline habitats, embayments, tributary mouths, and warm, shallow backwaters as habitat for larval and juvenile razorback suckers within ponds and reservoirs (Langhorst and Marsh 1986; Minckley et al. 1991, Pacey and Marsh 1998). The San Juan River flood plain does not have large, flooded bottomlands like the Green River system, suggesting that razorback sucker larvae can survive in the face of non-native 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 suckers 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 during the juvenile life stages of razorback suckers remains relatively understudied, as catches of juveniles remains minimal presumably due to the predatory and competitory impacts of non-native 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 only minimal data on juvenile habitat use available from the LCRB. Brandenburg et al. (2005) captured wild-spawned juvenile razorback suckers 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. 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 suckers that were released into Lakes Mohave and Powell. Their telemetry data demonstrated that juvenile razorback suckers used 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 suckers in the San Juan River (Holden 1999). These fish were found occupying slackwater and backwater pools 0.3 to 0.9 m deep and 1 to 3 °C warmer than adjacent main channel habitats. Albrecht et al. (2013) studied the habitat use of four sonic-tagged juvenile (sexually immature) razorback suckers in Lake Mead, finding that habitat use shifted seasonally. Lake Mead juvenile razorback suckers were observed using inshore habitat characterized by abundant vegetative cover, higher turbidity, and larger substrates during the spring months; offshore habitat characterized by greater depths, higher temperatures, and larger substrates in summer, and inshore habitat characterized by inundated vegetation, submerged aquatic vegetative cover, and silt substrates in fall. Razorback sucker studies in the LCRB on Lake Mead (Holden et al. 1997, 1999, 2000a, 2000b, 2001; Abate et al. 2002; Welker and Holden 2003, 2004) have documented the 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. Additional evidence of razorback sucker progression through all life stages in Lake Mead was documented again in 2013 with the capture of a wild 215-mm juvenile fish from the Colorado River inflow area of the lake (Kegerries and Albrecht 2013). This is the smallest razorback sucker captured to date in Lake Mead.

Studies by Modde (1996) and Modde et al. (2001) of the Green River demonstrate that juveniles and subadult razorback suckers may favor flood plain 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 of 26.6 °C) over main channel habitats based on habitat variables such as zooplankton density, water temperature, depth, and vegetation abundance. Laboratory studies have also demonstrated that warmer water temperatures (22–24 °C), such as those found in flood plain depressions, are preferred by juvenile razorback suckers (Bulkley et al. 1981; Valentine 1981). Modde (1996) and Modde et al. (2001) also indicate that growth and survival in flood plain depressions are more likely than in main channel habitats despite heavy impacts of non-native fish predation and competition typically associated with backwater habitats. Furthermore, they suggest that draining wetlands before spring may be important for non-native 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 flood plains), coupled with annual reset draining of backwaters (to remove residual non-native fishes), increases young razorback sucker growth and survival throughout the first year of life by allowing razorback suckers to exist at sizes similar to their non-native competitors and predators. Modde (1997) documents similar growth and survival rates for young-of-year razorback sucker and carp (*Cyprinus carpio*) in a managed wetland. Mueller et al. (2003) demonstrate that flow acclimation of stocked razorback suckers 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 suckers to lengths greater than 350 mm TL doubles post-stocking survival.

## Reproduction

Fecundity for razorback suckers expressed in terms of number of ova per unit standard length was derived by Minckley (1983). Estimates by Minckley (1983) ranged from 1,600 to 2,000 ova per centimeter standard length. These results were based on 15 fish estimated at having anywhere from a 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) indicates that during the spawning season in Lake Mohave, male:female ratios of razorback suckers range from 1.2 to 3.6:1. They also report that approximately 80% of male and less than 65% of female razorback suckers are ripe during peak spawning activity. A summary of sex ratios in Lake Mead, based on all wild razorback suckers captured from 1990 to 2013 for which sex could be positively determined, approximates a male:female ratio of 1:1.5 (B. Albrecht 2013, personal communication). A multi-year summary such as this may be more useful in reporting this information, as razorback suckers are a relatively long-lived species, and recapture data compiled by Albrecht and Holden (2005) from nine consecutive years of accumulated data indicate that adult female razorback suckers tagged with passive integrated transponder 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 suckers ranges from a minimum estimate of 2 years for male and 3 years for female razorback suckers to a maximum of 6 years for some populations, or it 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 was reported to occur in 5.2–5.5 days at 15 °C (Minckley and Gustafson 1982) and in 6–7 days at 18–20 °C (Snyder et al. 2004). Numerous other researchers have also evaluated the effects of incubation temperatures on razorback sucker eggs, finding that laboratory hatching rates are consistently the highest at 20–23.5 °C (Bozek et al. 1990; Haines 1995; Hamman 1985; Loudermilk 1981; Marsh 1985). Osmundson (2001) concludes that, based on available data, water temperatures near 20 °C during the spawning period are an important factor in the reproductive success of razorback suckers.

Egg mortality in river and reservoir settings has been attributed to fluctuating water levels, scouring by currents and/or wave action, suffocation due to silt deposition, and non-native 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). Razorback sucker larvae have been reported to swim-up in 12–13 days post-hatch at 18–20 °C and in 17–21 days post-hatch at 15 °C (Snyder et al. 2004). 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 the “Diet” section below). Due to the relationship between hatching, yolk absorption, and reaching the swim-up stage, it is important that larval razorback suckers occupy habitat with sufficient food sources within 1–2 days of swim-up to increase their likelihood of survival (Valdez and Nelson 2004). Larval razorback suckers are photosensitive and display diel patterns in drift periodicity (Carter et al. 1986; Burke 1995).

## Diet

The razorback sucker’s diet composition is highly dependent 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 the 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 suckers 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 per liter.

Later during growth (age and size information unknown, but at some point during the juvenile life stage), razorback suckers 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 suckers 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 suckers 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 suckers vary, and available information is highly dependent 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 suckers. The majority of growth information for larval and juvenile razorback suckers has been based largely on hatchery-produced fish (Brooks 1985; Marsh 1985; Marsh and Brooks 1989; Minckley et al. 1991; Mueller 1995). Razorback suckers 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 suckers appear to grow rapidly (e.g., growth of 55–307 mm in 6 months for young razorback suckers stocked into ponds) (Osmundson and Kaeding 1989), after which growth becomes minimal (2 mm per 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 (1996) studied the largest extant riverine population of razorback suckers in the Green River, Utah, where he also found very slow growth in adults (1.66 mm per year). The highest growth rates described, outside of isolated refugia and hatchery ponds, have been those reported for the Lake Mead razorback sucker population, with growth rates of adult fish approaching 10–20 mm per year (Holden 1999; Holden et al. 1997, 2000a, 2000b, 2001; Abate et al. 2002; Welker and Holden 2003, 2004). The average annual growth rate observed in Lake Mead in more recent years continues to be high, reported at 8 to 58 mm per year (Albrecht et al. 2007, 2008b, 2010c, 2013; Kegerries et al. 2009; Shattuck et al. 2011); however, it should be noted that this information is at times based on limited numbers of recaptured fish. 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 suckers 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 suckers 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 suckers (*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 suckers 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 suckers 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 suckers but that caution should be used in aging older and slower-growing fishes.

During the early years of 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 toward 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, it could be validated 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. From 1998 to 2012, 395 individual razorback suckers captured from Lake Mead have been aged from 2 to 36 years using this technique, with the majority of data being collected from wild naturally recruited fish (Albrecht et al. 2013). These data indicate regular, if not annual, recruitment of new, wild razorback suckers in Lake Mead – something that has not been documented anywhere else in the Colorado River basin in the recent past.

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# 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*. Yuma clapper rails (*R. l. yumanensis* Dickey) are one of four subspecies of the *obsoletus* group (Eddleman and Conway 1998). They were initially designated as a separate species, *R. 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). Clapper rails primarily inhabit salt marshes and mangrove swamps throughout their range; Yuma clapper rails inhabit freshwater marshes in the Southwestern United States and northern Mexico (Eddleman and Conway 1998; Hinojosa-Huerta et al. 2013). They are distinguished by paler, duller underparts and grayish edging of dorsal feathers. Their cheeks and postoculars are bluish or ashy gray (Eddleman and Conway 1998).

## LEGAL STATUS

Yuma clapper rails were listed as endangered on March 11, 1967, by the Secretary of the Interior pursuant to the Endangered Species Act of 1966 (U.S. Department of the Interior 1967). California originally listed Yuma clapper rails as endangered in 1971, relisted them as rare in 1978, and currently lists them as threatened (California Department of Fish and Wildlife 2013). In 1978, Arizona classified Yuma clapper rails as a species of special concern, similar to the Federal status of endangered (Arizona Game and Fish Department 2006). Nevada ranks Yuma clapper rails as S1: critically imperiled and especially vulnerable to extinction or extirpation due to extreme rarity, imminent threats, or other factors (Department of Conservation and Natural Resources, Nevada Natural Heritage Program 2008). They are listed as threatened in Mexico (Hinojosa-Huerta et al. 2013).

## DISTRIBUTION

### Historical Range

Grinnell (1914) did not encounter rails during an expedition/exploration from Needles, California, to Yuma, Arizona, in 1910 (Rosenberg et al. 1991). Yuma clapper rails were found along the lower Colorado River (LCR) after construction of dams and the subsequent creation of marsh habitat (Ohmart and Smith 1973). This subspecies 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 the 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

Along the LCR, this subspecies is primarily found in scattered marshes from the Colorado River Delta in Mexico, to Topock Marsh at the Havasu National Wildlife Refuge, near Needles, California (Wise-Gervais 2005; Hinojosa-Huerta et al. 2013). In previous years, the northern limit along the LCR was Laughlin Bay, Nevada (Rosenberg et al. 1991). This subspecies' 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). They are also found at the Ash Meadows National Wildlife Refuge (U.S. Fish and Wildlife Service [USFWS] 2011). Yuma clapper rails are 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 Department 2006). Significant populations are also found in marshes at the south end of the Salton Sea (Eddleman and Conway 1998; Patten et al. 2003, USFWS 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. 2008). Clapper rails present in mangrove marshes along the west coast of Mexico may also be Yuma clapper rails (Eddleman and Conway 1998).

## Populations Within the LCR MSCP Planning Area

Significant populations of Yuma clapper rails are found within the Lower Colorado River Multi-Species Conservation Program (LCR MSCP) planning area in Reaches 3–6. An analysis of survey data from 1995 to 2013 showed that between 30 and 58% of the Yuma clapper rails detected in the United States were within the LCR MSCP planning area (USFWS 2005, table 1; USFWS 2013, table 2). The majority of Yuma clapper rails located in Reach 3 were in Topock Marsh and Topock Gorge, and a small population was in the marshes of the Bill Williams River Delta. In Reach 4, the Cibola National Wildlife Refuge provides habitat for almost all Yuma clapper rails detected during surveys.

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

| Survey year | 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 | NS | NS | NS | NS | NS | NS | NS | NS | NS | NS | NS |
| Reach 3 | 83 | 86 | 76 | 85 | 56 | 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 |
| **U.S. total** | 900 | 834 | 814 | 579 | 543 | 503 | 533 | 639 | 851 | 863 | 885 |
| **LCR MSCP %** | 49.4 | 53.6 | 49.6 | 46.4 | 55.4 | 48.2 | 40.7 | 39.4 | 41.1 | 35.1 | 36.2 |

Table 2.—Yuma clapper rail/marsh bird survey data (USFWS 2013)
(NS indicates no survey.)

| Survey year | 2006 | 2007 | 2008 | 2009 | 2010 | 2011 | 2012 | 2013 |
|---|---|---|---|---|---|---|---|---|
| Reach 1 | NS | NS | NS | NS | NS | NS | NS | NS |
| Reach 2 | NS | NS | NS | NS | NS | NS | NS | NS |
| Reach 3 | 91 | 110 | 95 | 113 | 118 | 133 | 100 | 99 |
| Reach 4 | 42 | 33 | 17 | 34 | 6 | 52 | 24 | 31 |
| Reach 5 | 93 | 61 | 82 | 96 | 74 | 68 | 44 | 58 |
| Reach 6 | 74 | 68 | 45 | 83 | 64 | 76 | 68 | 55 |
| Reach 7 | 0 | NS | NS | NS | 0 | NS | 0 | NS |
| **Total** | 300 | 272 | 239 | 326 | 262 | 329 | 236 | 243 |
| **U.S. total** | 753 | 823 | 645 | 671 | 570 | 565 | 432 | 431 |
| **LCR MSCP %** | 39.8 | 33.0 | 30.1 | 48.6 | 46.0 | 58.2 | 54.6 | 56.4 |

In Reach 5, rails were detected in the Imperial National Wildlife Refuge, 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 planning area were: Reach 3: 31.6%, Reach 4: 16.8%; Reach 5: 25.3%; Reach 6: 25.7%; and Reach 7: 0.6%.

# LIFE HISTORY

## General Description

Yuma clapper rails are large, gray-brown to dull cinnamon rails, with slightly decurved bills, laterally compressed bodies, and long legs and toes relative to their body (Eddleman and Conway 1998). They are one of the smaller subspecies of clapper rails (Todd 1986). The total length for adult clapper rails is 32–41 centimeters (cm), with their mass ranging from 160 to 400 grams (g). Males are typically 20% larger than females (Eddleman and Conway 1998). Yuma clapper rail 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). Yuma clapper rails are the largest of the rails found along the LCR (Phillips et al. 1964). Large Yuma clapper rail males can stand 20 to 23 cm tall (Todd 1986). Virginia rails are similar in shape, but smaller at 22 to 27 cm in length, and have more red on their bills and more gray on their 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 toward 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 the 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 the Virginia rail (*R. limicola)* (Eddleman and Conway 1998; Peterson 1990). Todd (1986) and Eddleman (1989) suggest that molt patterns are similar between Yuma clapper rails and other clapper rails. Pre-juvenile 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 pre-juvenal molt. The definite pre-basic molt for adults is complete and occurs after breeding. During this time, remiges and rectrices are lost, and adult birds are flightless. The 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, while conducting surveys in Topock Gorge, Havasu National Wildlife Refuge, 177 rails were encountered audibly, with only 4 seen (Bureau of Reclamation 2005). Most of the calls were related to territoriality and breeding behavior (Massey and Zembal 1987).

Tomlinson and Todd (1973) describe seven calls for Yuma clapper rails: “kek,” “agitated kek,” “clatter,” “purr,” “agitated purr,” “hoo,” and “kek-burr.” An additional five vocalizations and calls were heard during a study along the LCR: “kek-hurrah,” “wheet,” “burp,” “kak,” and “rack” (Eddleman 1989). Tomlinson and Todd (1973) identify the “kek” and “clatter” calls of Yuma clapper rails as identical to those given by eastern clapper rails (*R. levipes* spp.). Massey and Zembal (1987) describe eight calls for the light-footed clapper rail (*R. l. levipes*). They state, “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 LCR 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 has 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." This 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 their 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

The age of Yuma clapper rails 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 March 13 (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 the first brood in southwest Arizona and southeast California is May 1 ± 24.8 days (Eddleman and Conway 1998). The 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. The 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 cattails (*Typha*) and salt cedar (*Tamarix* spp.) 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, or salt cedar; under wind-thrown bullwhip bulrush (*Juncus californicus*); and within or on top of dead cattails 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 the 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 Fancher 1988). Parental care extends to the fifth or sixth week (Adams and Quay 1958; Zembal and Fancher 1988), but brooding may continue until the 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; Williams1929; Meanly 1985; Todd 1986; Zembal and Fancher 1988). In Arizona, Yuma clapper rails forage 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 Yuma clapper rails 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, unidentified beetles, spiders, damselfly nymphs, grasshoppers, insect eggs, ground beetles, 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. Ohmart 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) observes Yuma clapper rails capturing small fish and tadpoles and gleaning invertebrates from algae-covered parts of cattails under the water surface. Eddleman (1989) looks at food habits and prey availability for Yuma clapper rails 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 salt cedar 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. 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, dragonfly nymphs, 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 (*Schoenoplectus tabernaemontani*), 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, it was determined through habitat studies 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 describe the ideal habitat as being a mosaic of emergent plant stands of different ages interspersed with shallow pools of open water.

Estimates of the home range of Yuma clapper rails at Mittry Lake were 24 hectares (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 were collected by attaching radio transmitters to rails and monitoring their movements during 1985–87 (Conway et al. 1993). Previous estimates of home range varied between 0.12 and 3.9 ha during their breeding season and are based on vocalizations, which may not be an effective method of determining Yuma clapper rail area use size (Todd 1986; Tomlinson and Todd 1973; Conway et al. 1993).

## Migration

Data are inconclusive concerning the migration of Yuma clapper rails. 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 winter, individuals do use the LCR all year. Changes in detection rates may influence broadcast call surveys conducted outside their 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 harriers (*Circus cyaneus*), great horned owls (*Bubo virginicus*), and Harris' hawks (*Parabuteo unicinctus*) have been documented as predators of Yuma clapper rails (Eddleman 1989). Striped skunks (*Mephitis mephitis*) are a potential predator of adult rails, and bullfrogs, black bass (*Micropterus salmoides*), softshell turtles (*Apalone spinifera*), and common king snakes (*Lampropeltis getulus*) are potential predators of young rails and eggs (Todd 1986).

Fires during their breeding season (mid-March to early September) can cause the loss of eggs, young, and some adults (Todd 1986). After breeding, adults go through a pre-basic 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 rail populations.

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 LCR, 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 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, such as dredging, channelization, bank stabilization, and other flood control measures, which are detrimental to marsh formation (Bureau of Reclamation 2004).

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# OTHER COVERED SPECIES

# Arizona Bell's vireo (*Vireo bellii arizonae*)

## LEGAL STATUS

The subspecies Arizona Bell's vireo (*Vireo belli 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. The California Department of Fish and Game listed the Arizona Bell's vireo as endangered in 1988.

## DISTRIBUTION

### Historic and Current Distribution Along the Lower Colorado River

Arizona Bell's vireos, until the 1950s, were quite abundant and much more widespread. Grinnell (1914) calls the Bell's vireo "one of the most characteristic avifaunal elements in the riparian strip." He estimates that one singing male occupied every 200 yards of willow (*Salix* spp.) habitat along the Colorado River's edge. Toward the middle of the century, Grinnell and Miller (1944) still classify this vireo as a common summer resident. By the early 1960s, Arizona Bell's vireos were 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). More recently, system-wide bird surveys have been conducted along the LCR, and the number of pairs detected has increased substantially.

Between 1976 and 1986, Rosenberg et al. (1991) estimates that the Arizona Bell's vireo population declined 57% along the LCR, with most of the reduction occurring after a flood event in 1983. From 1974 to 1984, breeding Arizona Bell's vireos occurred only in the vicinity of Needles, Topock Marsh, the Bill Williams River National Wildlife Refuge, and the Cibola National Wildlife Refuge, with a few scattered pairs near Parker and Poston, Arizona. A small population of the subspecies nested near the tip of Nevada in 1975 and 1976.

In 1981, 35 male Arizona Bell's vireos 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 kilometer south of the Agnes-Wilson Bridge (Serena 1986). Laymon and

Halterman (1986) found only four singing males during surveys of bird populations also on the California side of the LCR and concluded that Arizona Bell's vireos were on the verge of extirpation in California.

In 2007, the Great Basin Bird Observatory (GBBO) began conducting system-wide bird surveys along the LCR for the Lower Colorado River Multi-Species Conservation Program (LCR MSCP). These surveys have continued annually, with improvements to field methodology and the accuracy of estimation, through 2013 and are planned to continue to at least 2016. Substantially larger numbers of Arizona Bell's vireos have been found along the LCR using these surveys than were previously reported in earlier studies. The annual estimates of the number of Bell's vireo territories (pairs) along the LCR from 2011 to 2014 varied from 1,100 to 1,900 (GBBO 2012, 2013, 2014). It is unknown what has caused the large increase in the abundance of Arizona bell's vireo along the LCR.

## Current Range

Bell's vireos are widespread in the Southwestern and Central United States and northern Mexico (figure 1). Bell's vireos are 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, 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).

Arizona Bell's vireos breed in southeastern California to southwestern New Mexico and south to Sonora and Chihuahua, Mexico. The subspecies is currently an uncommon to locally common summer resident and breeder along the LCR (GBBO 2012, 2013, 2014).

## Populations Within the LCR MSCP Planning Area

Previously, a remnant population of Arizona Bell's vireos persisted on the Bill Williams River National Wildlife Refuge. In 1990, the Needles Municipal Golf Course supported patches of willow, mesquite (*Prosopis* spp.), tall cottonwoods (*Populus* spp.), and marshy ponds, which produced breeding Arizona Bell's vireos. Soto Ranch contained the last stand of mature honey mesquite (*Prosopis glandulosa*) on the California side of the LCR and supported Arizona Bell's vireos locally (Rosenberg et al. 1991).

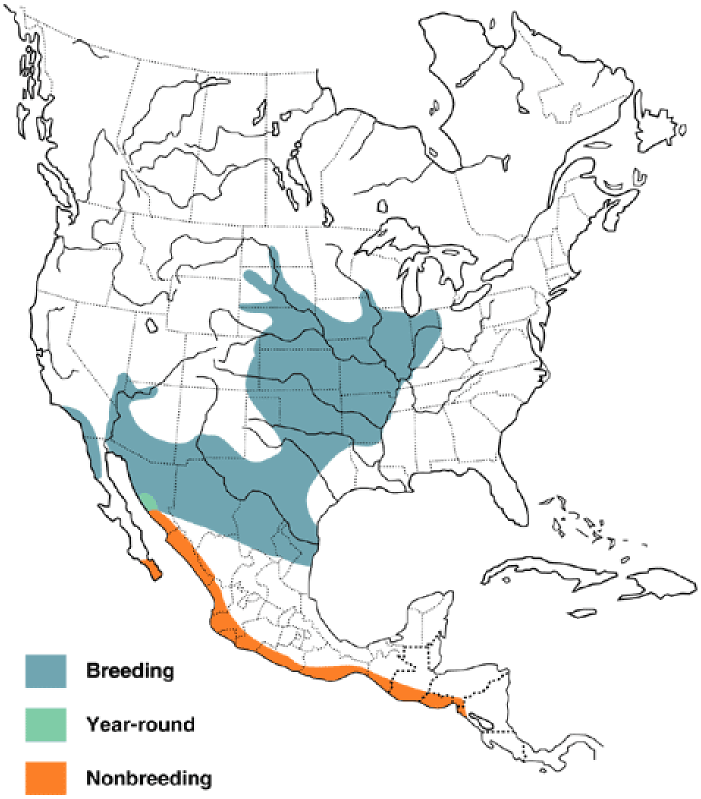

**Figure 1.—Bell's vireo wide breeding and wintering range.**

Currently, Bell's vireos are commonly found on the Bill Williams River National Wildlife Refuge and are also an uncommon summer resident along the rest of the LCR main stem. Resident, breeding Arizona Bell's vireos are present at two LCR MSCP restoration sites: Beal Lake Conservation Area and the Cibola National Wildlife Refuge Unit #1 Conservation Area.

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).

Previously, honey mesquite-salt cedar mixed stands represented the most important habitat of Arizona Bell's vireos, outside of the remaining willow habitats of the Bill Williams River National Wildlife Refuge and Needles, California (Rosenberg et al. 1991). Bell's vireos are found in areas of tall woody and low woody vegetation from Hoover Dam to the Imperial National Wildlife Refuge (GBBO 2011, 2012, 2013, 2014). The habitat designations are based on Bart et al. (2010) and are defined using the Anderson and Ohmart vegetation classification system (Andersen and Ohmart 1976). Tall woody is defined as consisting of cottonwood-willow I–III. Low woody is defined as consisting of cottonwood-willow IV–VI, salt cedar I–VI, honey mesquite III–VI, salt cedar-honey mesquite I–VI, and screwbean mesquite (*Prosopis pubescens*) and salt cedar I–VI.

# LIFE HISTORY

## General Description

The Bell's vireo is a small vireo, with a length of 115–125 millimeters and a weight of 7–10 grams. It 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

Arizona Bell's vireos are summer residents that generally breed between late March and late September. The earliest recorded arrival date at the LCR was March 8, and the latest departure date was from late November (Rosenberg et al. 1991). In Arizona, the peak period for breeding is from late April to late May, but dependent fledglings have been seen as late as September 19 (Corman and Wise-Gervais 2005).

According to Bent (1950), Arizona Bell's vireos are typical breeders of the streamside fringes of willows and mesquite along the LCR. Grinnell (1914) found these vireos 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-yard 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 meter (m) stretch, with large stretches of identical, unoccupied habitat among 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) reports 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 seepwillow *(Baccharis glutinosa)* 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 describes 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 x 6 centimeters (cm) in outside diameter and 4.5 cm in outside depth, and internally it is about 4.4 x 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).

Egg laying occurs from the end of April to the end of May. 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, although up to four broods can occur (Franzreb 1989). 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.

In a study on the detections of least Bell’s vireos (*V. b. pusillus*), it was found that detection probabilities declined during the month of May due to lower availability (Scott et al. 2005). Availability is the probability of a song occurring during any given minute. Availability may have been lower during the month of May due to nesting activities taking up time that may otherwise have been dedicated to singing.

Arizona Bell’s vireos are common victims of nest parasitism by brown-headed cowbirds (*Molothrus ater*) (Bent 1950). A female cowbird (*Molothrus* spp.) 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 nestling. 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 outcompetes 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. Sharp and Kus (2006) found that dense microhabitat cover is the most important factor in reducing the incidence of cowbird parasitism of least Bell’s vireos. It was also found that among pairs that were forced to desert a nest due to nest parasitism, the rate of nest success was greatly reduced even if the birds attempted to re-nest (Kus 2002).

## Feeding

Arizona Bell's vireos are summer visiting insectivores along the LCR (Rosenberg et al. 1991). Bent (1950) describes the subspecies as almost entirely insectivorous, and food items predominantly included various bugs, caterpillars, beetles, and grasshoppers. In one food habit study conducted during summer, the overall diet of Arizona Bell's vireos consisted of 34.4% true bugs and 18.5% 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, lady-bird beetles, weevils, leaf beetles, bees and wasps, spiders, snails, and, occasionally, wild fruit.

Grinnell (1914) reports that Bell's vireos forage in all vegetation associations but are most represented in the willow association, especially where there is an undergrowth of seepwillow. Bent reports that the subspecies works in a rather low foliage zone, from the ground to a height of 6–8 feet.

## Habitat

Early accounts indicate that Arizona Bell's vireos were most common in willow-dominated habitats, where they occupied the understory shrubs like seepwillow. Remnant LCR populations breed primarily in tall screwbean or honey mesquite woodlands near water, usually mixed with scattered willows and salt cedar. 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). Arizona Bell's vireos also breed 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 Arizona Bell's vireo breeding territories on the California side of the LCR. In all Arizona Bell's vireo territories measured, willows occurred in small patches and were interspersed with other plants. In work conducted through the Arizona Breeding Bird Atlas, the subspecies was found to use dense shrubby vegetation and woodland edges, especially mesquite, and can be found in drier habitats (Corman and Wise-Gervais 2005).

Bell's vireos are habitat generalists east of the LCR, with higher densities in honey mesquite and salt cedar 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. The subspecies is often, but not always, near water in arid regions. Vireos nest in any successional stage with dense understory vegetation, with the most critical structural component of the least Bell's vireo's 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 habitat used 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 least Bell's vireo 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 the least Bell's viero's historical range have been analyzed but not undertaken (Franzreb 1990). Currently, Arizona Bell's vireos have begun using restored habitat along the LCR without the need for translocation, and it does not appear necessary to use this technique.

## Threats

The near elimination of Arizona Bell's vireos as common breeding residents along the LCR was 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). Arizona Bell's vireos are frequent hosts to brown-headed cowbirds along the LCR (Serena 1986; Rosenberg et al. 1991). More recently, numbers of Arizona Bell's vireos have increased along the LCR, and the immediate threat of extirpation in the region is no longer a concern. As restoration efforts continue along the LCR and more mature mesquite habitat develops, there may be an increase in suitable habitat for Arizona Bell's vireos. Research on the use of restored habitat by the subspecies continues at LCR MSCP restoration sites and will inform future restoration efforts and the status of this subspecies along the LCR.

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# California Black Rail (*Laterallus jamaicensis coturniculus*)

## INTRODUCTION

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

## LEGAL STATUS

The U.S. Fish and Wildlife Service (USFWS) lists the California black rail as a bird of conservation concern (USFWS 2008). In Arizona, it is listed as a wildlife species of special concern (Arizona Game and Fish Department 2002). In California, this subspecies is listed as threatened (California Department of Fish and Wildlife 2013). The California black rail is listed as endangered in Mexico (Hinojosa-Huerta et al. 2013). It is not listed in Nevada.

## DISTRIBUTION

### Historical Range

Bent (1926) lists the range of California black rails as the Pacific coast of California. Grinnell (1915) states that black rails were fairly common fall and winter visitors 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. California black rails were common summer residents and breeders in the salt marshes of San Diego Bay (Grinnell 1915). They were found nesting inland at Chino, San Bernardino County, in 1935 (Hanna 1935) and observed at Riverside in 1893 and San Bernardino in 1919, both in the month of August (Wall 1919). In Arizona, black rails were listed as hypothetical (Swarth 1914; Phillips et al. 1964). Black rail were listed as very rare migrants in the Tacoma, Washington, area (Bowles 1906). They were also found inland in northern Baja California, Mexico (Huey 1928). There are no historical records for black rails 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 black rails along the lower Colorado River (LCR) were in June through August 1969. Four to six rails were observed near Imperial Dam (Snider 1969); the subspecies was 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 black rails at the Bill Williams River Delta was April 1978, and a population of 14 was 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 was found during surveys along the All-American Canal, west of Yuma, in 1984 (Kasprzyk et al. 1987). Individual black rails were observed at the Henderson Bird Viewing Preserve 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 along the Colorado River Delta, Baja California, and Sonora, Mexico (Hinojosa-Huerta 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 their breeding season, California black rails are 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 (Manolis 1978). Further to the south, rails are 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 LCR, California black rails are found at the Cienega de Santa Clara and several other sites in the Colorado River Delta in Mexico (Hinojosa-Huerta et al. 2013). They are found from Laguna Dam north to Imperial Reservoir, in portions of the Imperial National Wildlife Refuge, on Colorado River Indian Tribe lands, in the Bill Williams River National Wildlife Refuge, and in the Havasu National Wildlife Refuge (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 California black rails 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; Manolis 1978; Flores and Eddleman 1991; Rosenberg et al. 1991). They are now casual winter visitors along the coast of southern California, where they once bred (Garrett and Dunn 1981).

## Populations Within the LCR MSCP Planning Area

At present, there are no known populations of California black rails within Reaches 1 or 2. In Reach 3, the Bill Williams River Delta is a known location; the USFWS lists black rails as residents in the Bill Williams River National Wildlife Refuge (Rosenberg et al. 1991; USFWS 2012). California black rails have been encountered in Topock Gorge during marsh bird surveys, starting in 2007 (USFWS 2014). 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 Lower Colorado River Multi-Species Conservation Program (LCR MSCP) planning area. In Reach 5, rails are found between Imperial Dam and Martinez Lake, at Ferguson Lake, and at managed wetlands on the Imperial National Wildlife Refuge (Evens et al. 1991; USFWS 1994; Conway et al. 2002). In Reach 6, California black rails are 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 centimeters (cm), a wingspan of 22–28 cm, and a mean mass of 29–35 grams. California black rails are 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 wings are shades of dark gray, sometimes with a chestnut or brown wash, and scattered with white spots. The amount of spotting varies among individuals. The tail feathers are brownish gray. 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 California black rails is sexually dimorphic; the throats of females are pale gray to white, and the ventrums are medium to pale gray, whereas males are darker, with pale to medium gray throats. The sexes are similar in size (Eddleman et al. 1994). The bill of the black rail is short and black (Sibley 2000). The lengths of the tarsi are approximately 2.5 cm, and the colors of the tarsi and toes are grayish brown (Flores and Eddleman 1991; Eddleman et al. 1994). 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 LCR 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 LCR 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 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).

Black rails are very secretive and seldom seen; they run swiftly and quickly on the ground, are generally reluctant to fly, and their short flight is typical of rails (Eddleman et al. 1994). Their flight can be fast and strong over long distances (McMullen 1944 *in* Eddleman et al. 1994). They can also swim for short distances (Weske 1969).

Black rails lose their flight and tail feathers after breeding, usually between July 1 and August 31, and are 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). California black rails 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 (*Eleocharis* spp.). These nests were elevated above the mud substrate in clumps of vegetation: three in California or giant bulrush (*Scirpus californicus*), one in southern cattail, and one in three-square bulrush (*Scirpus 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 California black rails lay 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 three to seven eggs; this is similar to clutch sizes reported from California (four to eight eggs) (Bent 1926). Both sexes incubate the eggs, and the incubation period for the nests in Arizona ranged from 17 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 black rails are poorly known (Flores and Eddleman 1991). Small aquatic and terrestrial invertebrates of less than 1 cm and seeds are the main food items (Eddleman et al. 1994). Black rails in Arizona were found to consume predaceous diving beetles, ground beetles, other beetles, earwigs, and the seeds of Olney bulrush (*Scirpus olneyi*), California bulrush (*Schoenoplectus californicus*), and southern cattail during their breeding season. In late summer and autumn, their diet included grasshoppers, beetles, ants, earwigs, spiders, snails, 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). Black rails are probably daytime feeders and are active throughout the day (Weske 1969 *in* Eddleman et al. 1994; Flores 1991).

## Habitat

California black rails inhabit tidal marshes and freshwater marshes in the Western United States and Mexico (Eddleman et al. 1994; Hinojosa-Huerta et al. 2001). They use sites with shallower water than other North American rails (Eddleman et al. 1988). California black rails inhabit the drier portions of wetlands (Flores and Eddleman 1991). Inland sites, such as those along the LCR, are characterized by shallow, stable water levels; gently sloping shorelines; and vegetation dominated by fine-stemmed bulrush (*Scirpus* spp.) or grasses (Repking and Ohmart 1977). Todd (1977) states that black rails use dense stands of three-square bulrush along the LCR. Three-square bulrush is restricted to shallow water or moist soil (Conway et al. 2002). Flores (1991) describes microhabitats of black rails 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. This subspecies selects 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 choose marsh edges with a water depth less than 2.5 cm 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 (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). The subspecies is also associated with plants of the upland/wetland interface such as seep willow (*Baccharis glutinosa*), arrowweed (*Tessaria sericea*), saltgrass (*Distichlis spicata*), and cottonwood (*Populus* spp.) (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 hectares (ha) during the whole year. For all seasons, there were no significant differences in home ranges 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, the home ranges of different males seldom overlapped except during 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 California black rails include great egrets (*Casmerodius albus*), great blue herons (*Ardea herodias*), ring-billed gulls (*Larus delawarensis*), northern harriers (*Circus cyaneus*), short-eared owls (*Asio flammeus*), and possibly loggerhead shrikes (*Lanius ludovicianus*) (Huey 1926; Ewan 1928; Evens and Page 1986). There has been at least one report of a domestic cat capturing a black rail, 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 habitats by high tides are vulnerable to predation if they are unable to secure upland cover (Evens and Page 1986). This may be a possible scenario in managed wetlands along the LCR.

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 the 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 LCR (Conway et al. 2002).

Selenium may be a threat to California black rails along the LCR. Liver samples from four adult rails and one egg were analyzed for this element. 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 black rails are unknown (Eddleman et al. 1994).

Adult black rails lose their flight and tail feathers during their definite pre-basic 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.

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# Colorado River Cotton Rat (*Sigmodon arizonae plenus*)

## INTRODUCTION

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

## LEGAL STATUS

Colorado River cotton rats are federally listed as a Candidate 2 species and as a species of special concern in California (Blood 1998).  They are also classified as an endangered species by the International Union for Conservation of Nature (Blood 1998).

## DISTRIBUTION

Arizona cotton rats have a current distribution that includes southeastern California, Arizona, and western Mexico, and they historically extended up to extreme southern Nevada (Hoffmeister 1986).  The exact current and historical distributional range of Colorado River cotton rats is not well known.  According to data collected in 1934, the first records of *Sigmodon* spp. in Nevada came from a marsh in the extreme southern portion of the State (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).  In 2011, a Colorado River cotton rat was captured in the Big Bend Conservation Area near Laughlin, Nevada.  This marks the first species encounter in Nevada since 1961.  Trapping events through 2013 have confirmed a resident population in the area (Hill 2012).

Further south, records of Colorado River cotton rats are more common. Individuals have been documented in Arizona from Parker to Ehrenburg (Hoffmeister 1986). In California, Goldman (1928) reports the subspecies 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 Colorado River cotton rats 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 (1999) conducted trapping for a variety of small mammal species at two sites along the Colorado River, and at each site, several Colorado River cotton rats were captured. The survey sites included a site recently revegetated on the Cibola National Wildlife Refuge and No-Name Lake near Parker, Arizona. All sites were located on the eastern (Arizona) side of the river. Studies suggest that, within their area of occurrence, the presence of Colorado River cotton rats is isolated and spotty rather than continuous.

The southern extent of the range of Colorado River cotton rats is still unknown. In work conducted by Blood (1990), a qualitative analysis found this subspecies occurring north of the Palo Verde Mountains, along the lower Colorado River (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) hypothesizes that, in some areas, Colorado River cotton rats may become sympatric with Yuma hispid cotton rats, and given the possible expansion of both into agricultural areas, this may be now occurring. However, a recent study of both rats along the LCR has produced no evidence that their ranges overlap (Neiswenter 2010). In Neiswenter (2010), Yuma hispid cotton rats were not captured north of the Imperial National Wildlife Refuge, and Colorado River cotton rats were not captured south of the Cibola National Wildlife Refuge. In addition to the distribution ranges for *Sigmodon* spp. from the Neiswenter (2010) study, genetic analyses determined two different unique haplotypes of Colorado River cotton rats were found along the LCR and a single individual with a haplotype found in eastern Arizona. These data give further evidence that the *plenus* subspecies is mostly disjunct from other subspecies in Arizona.

Additional surveys for Colorado River cotton rats have confirmed their presence on the Havasu National Wildlife Refuge near Pintail Slough and on the Big Bend Conservation Area, within the Palo Verde Ecological Reserve just north of Blythe, California; within the Cibola Valley Conservation Area; and in the Cibola National Wildlife Refuge Unit 1 Conservation Area (Hill 2011, 2012).

# LIFE HISTORY

## General Description

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

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

## Identification

Identification of Colorado River cotton rats is very difficult due to their possibly overlapping distribution with Yuma hispid cotton rats 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 Colorado River cotton rats from Yuma hispid cotton rats (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), which would suggest that hybridization is rare, if it occurs at all.

## Breeding

No specific information exists on the breeding habits of Colorado River cotton rats, but they are 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 Arizona cotton rats have been collected during almost every month of the year (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 [n.d.]).

## Habitat

In capture studies conducted by Andersen and Nelson (1999), Colorado River cotton rats were most readily captured in grass/cattail (*Typha*) communities. In restoration sites, this subspecies was not captured until an herbaceous understory had developed. Andersen and Nelson (1999) conclude that the development of an understory may be the most important element in providing quality habitat for many species of small mammals, including Colorado river cotton rats. In preliminary presence/absence studies conducted by the Bureau of Reclamation at the Cibola National Wildlife Refuge Nature Trail restoration site on the Cibola National Wildlife Refuge in 2004–05, Colorado river cotton rats were only captured in areas with a dense understory of Johnsongrass (*Sorghum halapense*) (Dodge 2006).

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

In a recent habitat modeling study, it was found that the two most important variables in the Colorado River cotton rat's survivorship were vegetation vertical density at 10–20 centimeters and 90–100 centimeters (Neiswenter 2011). These two variables are the greatest predictors of Colorado cotton rat presence. The study identified native *Chlorocantha spinosa* as having the ability to provide habitat structural components identified by the habitat models.

## Threats

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

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# Elf Owl (*Micrathene whitneyi*)

## LEGAL STATUS

Elf owls (*Micrathene whitneyi*) are not a federally listed species. California listed them as endangered in 1980 (California Department of Fish and Wildlife 2013). Elf owls are not listed in 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). Fossil records from 11,000–27,000 years ago were also discovered in Nuevo León, Mexico (Steadman et al. 1994). Fossil records from central New Mexico are known from 10,795 ± 50 years (Brasso and Emslie 2006). Henry and Gehlbach (1999) suggest that elf owls occupied evergreen woodlands and riparian forests in the present Sonoran Desert region before saguaro (*Carnegiea gigantea*) cacti 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 (LCR), 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

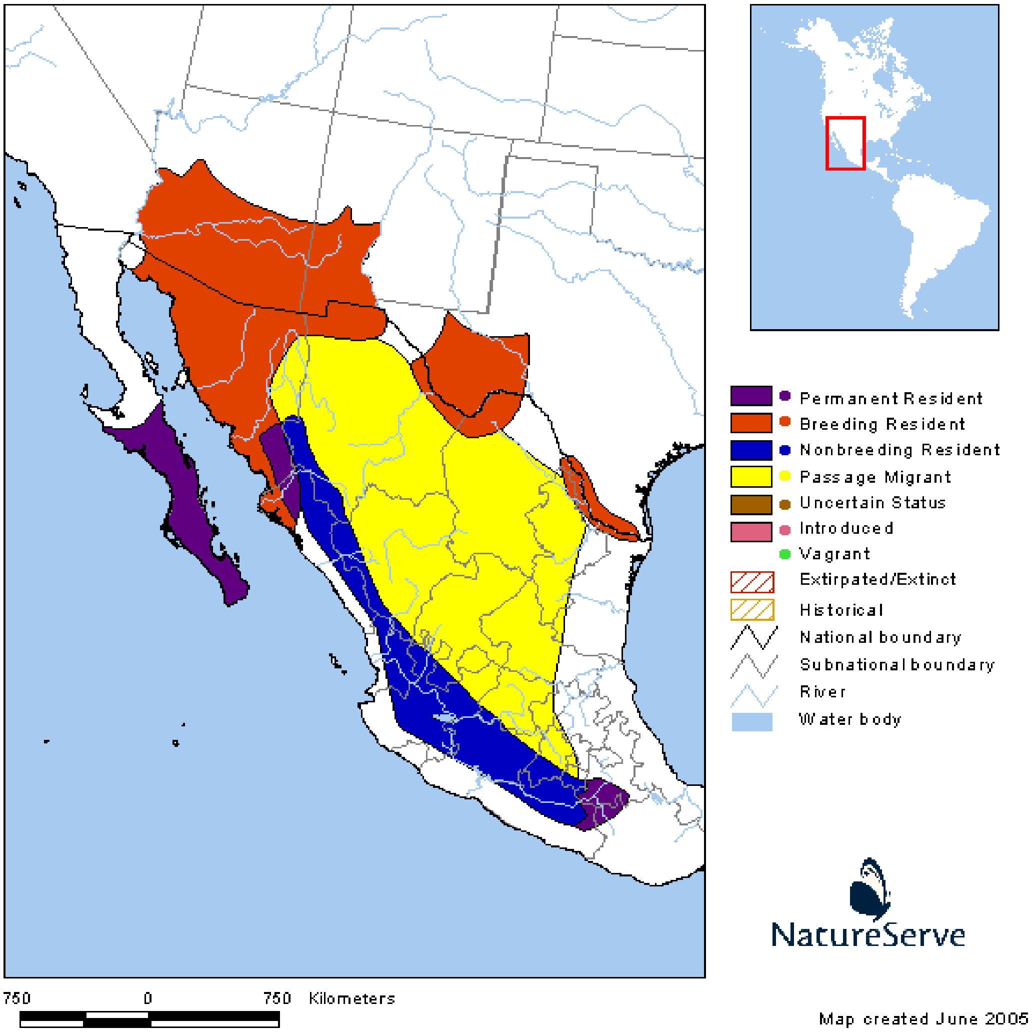

Figure 1.–Elf owl distribution.

In Mexico, there are three distinct year-round and wintering populations of elf owls (figure 1), including:

1. The southern portion of Baja California, which supports a year-round population
2. Socorro and other Revilla Gigedo Islands, which support a year-round population
3. A wintering population located from southwestern Puebla to northern Oaxaca

## Populations Within the LCR MSCP Planning Area

In the past 100 years, elf owl presence in the Lower Colorado River Multi-Species Conservation Program (LCR MSCP) planning 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 (1903, 1904) reports observing elf owls near Yuma, Arizona, and in Duncan Flats and Senator Mine in California opposite Yuma. Stephens (1903) observed an elf owl near Needles, California, on the Arizona side of the river. Kimball (1922) reports elf owls using cottonwood (*Populus* spp.) trees in Bard, California. Elf owls were also reported in the Kofa Mountains east of Yuma (Walker 1943). Miller (1946) reports 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. The higher numbers from Halterman et al. (1987) may be attributed to the fact that they surveyed almost twice as many sites as Cardiff (1978, 1979). McKernan and Braden (2002) report 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 (*Empidonax traillii extimus*)surveys. Data from the Arizona Breeding Bird Atlas (Wise-Gervais 2005) describe elf owls at the Bill Williams River Delta. Table 1 summarizes elf owl observations along the LCR.

The California Department of Fish and Wildlife (CDFW) conducted surveys from 1998 to 2000 at all sites surveyed in the 1970s and 1980s, and no elf owls were detected (California Department of Fish and Game [CDFG] 2005). An elf owl was heard at Marcus Wash at Picacho State Recreation Area in 2000 (McKernan and Braden 2002; P. Jorgenson 2008, personal communication). An elf owl was heard near Soto Ranch in 2002 (J. Sterling 2008, personal communication; Bureau of Reclamation [Reclamation] 2004b; CDFG 2005). In 2012, an elf owl pair was breeding at Corn Springs, California (J. Boone 2012, personal communication).

Reclamation conducted surveys at selected sites from 2008 to 2010, and one elf owl was detected near Blankenship Bend on the Arizona side of the Havasu National Wildlife Refuge (Sabin 2009, 2010).

Table 1.—Elf owl observations along the LCR

| Area | Elf owls observed | Citation |
|---|---|---|
| Needles, California | 3+ | Stephens 1903 |
| Needles, California-Yuma, Arizona | 20, 10,<br>30–36 | Cardiff 1978, 1979,<br>Halterman et al. 1987 |
| Needles, California | 1–2 | Bureau of Reclamation 2004b, CDFG 2005 |
| Havasu National Wildlife Refuge, Arizona Blankenship Bend | 1 | Sabin 2009, 2010 |
| Joshua Tree National Park, Riverside County, California | 2 | Miller 1946 |
| Lake Havasu south to Taylor Lake | Unknown | McKernan and Braden 2002 |
| Bill Williams River Delta, Arizona | Unknown | Wise-Gervais 2005 |
| Kofa Mountains, Yuma, Arizona | 5 | Walker 1943 |
| Yuma, Arizona | 5, 5 | Brown 1903, 1904 |
| Bard, California | 2 | Kimball 1922 |
| Corn Springs, California | 1 | J. Boone 2012, personal communication |

# LIFE HISTORY

## General Description

The elf owl is the smallest owl in the world, with adults measuring 12.4–14.2 centimeters (cm) in length and weighing 35–55 grams.  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.  The elf owl has a short tail with a buffy band.  The sexes are alike in plumage, with the female unnoticeably larger.  The plumage remains the same throughout the year.  Four months after hatching, juvenile plumage is mottled grayish brown and resembles that of 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

A developing nestling makes a soft peep or squeak, and a twitter followed by a rasp, approximately 48 times per minute, with the volume proportional to hunger (Gilman 1909; Ligon 1968).  Hardy et al. (1999) report hearing the rasps up to

100 meters 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 five to seven notes, delivered at the rate of five to six notes per second, with a faint beginning note rising with emphasis at the end. Their song resembles a dog yipping. Elf owls perform these chatter songs as a nest site proclamation/defense and as advertisements by males to females. Chatter songs vary in frequency and become more insistent when humans or conspecifics are near. Another version of the chatter song is eight to nine notes per second, lasting more than 1 minute, increasing and decreasing with volume. Males perform this song for females as intense nest cavity advertisement. In flight, males advertise to 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 trill of nestlings (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 spring during pair formation and through 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. Songs 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 their breeding range, elf owls are associated with mature saguaro cacti (*Carnegiea gigantae*) (Stephens 1903; Campbell 1934). They have been associated with riparian forests, desert wash woodlands, upland deserts, canyon riparian forests, dry oak woodlands, wooded canyons, sycamores, and evergreen woodland habitats (CDFG 2005; Wise-Gervais 2005). 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; Cardiff 1978, 1979; Halterman et al. 1987). Optimal habitat along the LCR has been described as areas that contain: (1) mature stands of cottonwood, willow, and mesquite trees, (2) a number of excavated cavities that serve as potential nesting sites, (3) large patch size, (4) high composition of native habitat and low composition of non-native habitat, and (5) a high percentage of canopy cover (Cardiff 1978; Halterman et al. 1987). However, this information on optimal habitat is based on observing elf owls while conducting presence/absence surveys and not quantitative habitat research.

There have only been a few studies on the habitat associations of elf owls in select areas and habitat types within their range, and from those studies the following information can be gleaned. Elf owls seem to be associated with a large number of excavated cavities (Hardy and Morrison 2001; Wise-Gervais 2005). Hardy and Morrison (2001) found that the number of potential nest cavities in a saguaro was the most important factor in determining if it was used for nesting. In saguaro habitat, elf owl abundance was positively correlated with the density of larger and more structurally complex saguaros that usually contain more cavities (Goad and Mannon 1987; Hardy et al. 1999). An abundance of cavities in a territory may provide roost locations and alternate cavities in case of nest failure or usurpation (Hardy et al. 1999). Hardy and Morrison (2001) found that elf owls prefer cavities higher off the ground, cavities in the main stem of the saguaro, and north-facing cavities. The owls may have been selecting cavities that provided a thermoregulatory advantage (Hardy and Morrison 2001; CDFG 2005).

The presence of mesquite and an understory and overstory layer of perennial vegetation may be important to elf owls, but more research would be beneficial to understand their habitat requirements in riparian habitat (Hardy et al. 1999; Hardy and Morrison 2001). Gamel and Brush (2001) found that elf owls select habitats with a distinct partial canopy layer at 4 meters in height and a semiopen understory where it is dense at 2.5 to 3 meters. Gamel and Brush (2001) found that elf owls do not use areas with very dense or no understory and a very high or low canopy. Hardy (1997) found that ocotillos and ironwoods are denser in front of the saguaro nest tree; owls may be using the tall trees as food delivery perches.

During their non-breeding season, elf owls use tropical deciduous forests with columnar cacti (*Stenocereus* spp. and *Neobuxbaumia* spp.) and arid grassy savanna with columnar cacti (*Lemaireocereus* spp.) (Henry and Gehlbach 1999).

Very little habitat research has been done regarding elf owl habitat associations in cottonwood-willow and mesquite lowland riparian areas. Under the LCR MSCP, studies are being coordinated that will provide more quantitative information regarding elf owl habitat associations within lowland riparian areas. The first phase of research will involve studying how often and for what purposes elf owls are using the cottonwood-willow and mesquite habitat within their range.

# Diet

The major food items of elf owls are arthropods from 22 families, consisting principally of insects, including moths, beetles, and crickets (Henry and Gehlbach 1999; CDFG 2005). They have also been known to eat other arthropods such as centipedes and scorpions (CDFG 2005). Their prey type changes to primarily scarab beetles as the summer rains bring large population of these insects (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 with their beaks or feet via a sit and wait strategy from strategic perches. Occasionally, they run after their prey on the ground. Elf owls probe for insects on flowers (Walker 1943; Marshall, Jr. 1957; Ligon 1968). Ligon (1968) reports elf owls removing scorpion stingers and dangerous terminal abdominal ends in order to avoid poisonous parts.

# Behavior

Adult flight of elf owls 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 known about elf owl self-maintenance, whereas preening and allopreening are widely reported in most other owls (Forsman and Wright 1979).

To avoid detection, elf owls stand erect and compressed, with one wing partially covering their face, and their side toward the intruder, following the intruder’s movement. When threatened, elf owls erect their breast contour feathers, lower one or both wings slightly, sway their body, and clap their bill (Ligon 1968). Nesting females have feigned death when approached by humans (Brown 1903; Ligon 1968).

The home range of elf owls is from 0.2 to 0.4 hectares, 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 their territory against those from adjacent breeding territories, but there does not seem to be competition within family groups nor between non-breeding individuals, which suggests there is no intraspecies hierarchy. Ligon (1968) 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 their breeding grounds in March and leave in September (Phillips 1942; Phillips et al. 1964) and spend their non-breeding season in Mexico as far south as Oaxaca. Ligon (1968) suggests that elf owls migrate because arthropod food resources are more active farther south during winter.

## Threats

Great horned owls (*Bubo virginicus*) and Cooper's hawks (*A. cooperii*) prey 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

Elf owls are secondary cavity nesters, relying on cavities excavated by other birds. 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, which 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 meters, and the range is from 3 to 11 meters (Goad and Mannan 1987). In the mountain canyon of southeastern Arizona, nest heights averaged 10 meters, with a range of 5–18 meters in sycamore trees (Ligon 1968). Nest cavity microclimate is more stable than the outside ambient temperature (Ligon 1968; Henry and Gehlbach 1999). Goad and Mannon (1987) found that elf owls have no preference for the size of the cavity; however, Hardy and Morrison (2001) found that elf owls prefer Gila woodpecker (*Melanerpes uropygialis*) cavities over the larger gilded flicker (*Colaptes Chrysoides*) cavities.

Elf owls have one brood and replace lost clutches (Ligon 1968; Henry and Gehlbach 1999). They lays 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 one to two 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) and 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 grams 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; thus, 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 100%.  Fledgling success is 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 among habitat types, from 2.2 to 5.0 pairs per square kilometer.  The lower densities occur at the edges of 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 of rainfall), nest box use declined 6%, but productivity decreased 52% (McKinney 1996).

## Survey Protocol

Historically, call-broadcast methods in which elf owl calls are broadcasted at a certain decibel to elicit responses have been used when conducting presence/absence surveys and habitat research.  Boal and Bibles (2001) demonstrate that elf owl responses increase when call-broadcast methods are used versus passive surveys.  However, the distance in between the broadcasts, the length of broadcasts, and the decibel level the broadcasts were played at varied among studies and survey efforts (Cardiff 1978, 1979; Gamel 1997; Hardy et al. 1999; Boal and Bibles 2001; Stiedl 2001, 2002).  Elf owl calls can be detected at further distances in open upland habitat versus the more dense riparian habitat (Great Basin Bird Observatory [GBBO] 2012).

LCR MSCP staff coordinated a detectability study on elf owls that provided quantitative responsiveness estimates to different time periods throughout the night, time periods throughout their breeding season, and distance categories (GBBO 2012).  The time from when the broadcast was played and the owl first responded was also recorded.  From this information, a standardized protocol was created (GBBO 2012).  Further detectability testing is currently being coordinated under the LCR MSCP to refine the protocol (GBBO 2012).

# 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 elf owls. 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 is not known. Researchers and birders may have a negative effect on them 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 they would use 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, elf owls were using 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.

Competition between elf owls and other cavity-nesting species, such as European starlings (*Sturnus vulgaris*), need to be quantified and remedial measures, if necessary, determined. Demographic data need to be collected, which may include items such as fledgling dispersal, prey abundance, home range size, and cavity selection.

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# Flannelmouth Sucker (*Catostomus latipinnis*)

## DISTRIBUTION

Flannelmouth suckers (*Catostomus latipinnis*) were 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, they were found to be declining in the Lower Colorado River Basin (LCRB), and for a period, they were largely restricted to the Colorado River above Lake Mead and a short segment of the Salt River (Minckley 1973). Flannelmouth suckers apparently are not able to persist in large impoundments, unlike razorback suckers (*Xyrauchen texanus*). Jonez and Sumner (1954) found razorback suckers to be relatively common in Lakes Mead and Mohave, while flannelmouth suckers were only encountered rarely. This trend appears to continue, with annual catches of razorback suckers 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, Inc., unpublished data), and only two flannelmouth suckers have been captured in Lake Havasu (Mueller and Marsh 2002). Interestingly, flannelmouth suckers may never have been vastly abundant in the LCRB. For example, Dill (1944) did not indicate the presence of flannelmouth suckers in the LCRB during his sampling efforts at the turn of the century. Miller (1961) reported few captures of the species in the LCRB, but he stated that the species was found in the Salt and San Pedro Rivers in the early 1900s. Furthermore, flannelmouth suckers were 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 suckers originating from the Paria River into the main stem 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) reported that by the mid-1990s, young flannelmouth suckers 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 suckers is of particular interest because similar trends in flannelmouth sucker abundance and distribution were noted following disturbances from the construction of main stem dams in the UCRB. For example, Wiltzius (1976) indicates that flannelmouth suckers are being replaced by introduced longnose suckers, a phenomenon likely attributable to the cold-water releases from mainstream dams and competitive interactions with non-native species. Flannelmouth suckers occupy approximately 45% of their historical range in the Colorado River Basin – 50% in the UCRB and 25% in the LCRB (Bezzerides and Bestgen 2002). However, flannelmouth suckers remain relatively abundant in the UCRB in less-altered sections of the river (McAda 1977; McAda and Wydoski 1985; Tyus et al. 1982) and in the Colorado River inflow of Lake Mead (Keggerries and Albrecht 2013); they are the most abundant large-bodied fish species in some river sections (Ryden 2005, 2012).

As discussed below, flannelmouth suckers are generalists of the river, especially with regard to nursery habitat requirements, compared with the other rare species (razorback suckers, bonytail, pikeminnows, 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 suckers and other large-river fishes has been the construction of main stem 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 non-native fishes that are successfully established in the Colorado River and its reservoirs have also contributed to their decline (Minckley and Deacon 1991; Johnson et al. 2008; Karam and Marsh 2010).

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, see Tyus et al. (1982), Minckley and Deacon (1991), Chart and Bergersen (1992), and Mueller and Marsh (2002).

## SYSTEMATICS AND MORPHOMETRICS

Flannelmouth suckers belong 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 suckers are distinguished from other species of the genus *Catostomus* by the thickened lower lip, which 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 suckers as typically dark brownish-green dorsally, yellowish or orange laterally, and white ventrally. However, in more turbid reaches, flannelmouth suckers 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 suckers reach lengths over 18 inches (Eddy and Underhill 1969), with some of the largest specimens being captured in the LCRB, where lengths can exceed 26 inches (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 suckers, hybridization between flannelmouth suckers and other native Colorado River catostomid species has historically been documented to occur. Flannelmouth suckers have been shown to hybridize with populations of razorback suckers. It is foreseeable that populations of flannelmouth suckers would hybridize with Sonora suckers (*Catostomus insignis*) and other native catostomids given that flannelmouth suckers 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) use allozymic data to directly quantify presumed introgression in the range of their samples as being 0–5% toward flannelmouth suckers and 0–3% toward razorback suckers. Hybridization between flannelmouth and razorback suckers is problematic given that adults of both species have been documented to simultaneously use cobble-bottomed, main channel riffles for spawning activities in natural riverine settings (see the "Razorback Sucker [*Xyrauchen texanus*]" species profile in this document). A larger concern currently is hybridization between flannelmouth suckers and introduced sucker species. Hybridization has been documented between flannelmouth and white suckers in most UCRB streams (Holden 1973; Ryden 2005). Currently, there are no known non-native sucker species in the LCRB.

# Habitat

## Adults

Historically, flannelmouth suckers inhabited virtually all components of riverine habitat ranging from fast current, to riffle, eddy, and stagnant backwater areas. Flannelmouth suckers have been known to be the predominate species comprising native fish catches (Holden 1973; Holden and Stalnaker 1975; McAda 1977; Holden 1999). Minckley (1973) describes flannelmouth suckers as being inhabitants of the larger, swifter streams and rivers of the Colorado River Basin. Cross (1975) found flannelmouth suckers 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 suckers 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). Similar observations were documented in the LCRB by Best and Lantow (2012), with over 60% of telemetered detections occurring within cobble substrates. Flannelmouth suckers also appear to have an 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 suckers were typically found in deeper water (mean 59 ± 49 centimeters in the Virgin River), and water velocities at capture were variable (0–1.0 meter per second, mean value of 0.44 meter per second) (Cross 1975). More applicable to main stem Colorado River habitats, Gaufin et al. (1960) found adult flannelmouth suckers 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 suckers 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 meters 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 suckers.

Chart and Bergersen (1992) document the migrational impacts on flannelmouth suckers 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 suckers to their home ranges after dam closure, indicating the likelihood of large, seasonal, migrational movements historically. Furthermore, Vanicek et al. (1970) found flannelmouth and bluehead suckers (*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 suckers (Holden 1999; Mueller and Wydoski 2004). Radio telemetry work by Beyers et al. (2001)

suggest that habitat use by adult flannelmouth suckers does not change with the time of day, unlike other native fishes that tend to display unique diel habitat use patterns (see the “Bonytail [*Gila elegans*]” species profile).

Furthermore, flannelmouth suckers tracked by telemetry were found in water ranging from 0.5 to greater than 6.0 meters in depth, and during the spawning season, most contacts were made in depths between 1–2 meters (Best and Lantow 2012). Cross (1975) mentioned that flannelmouth suckers 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 flannelmouth suckers are not as specific in their habitat selection, compared with other large-bodied native fishes (e.g., razorback suckers, bonytail, humpback chubs [*Gila cypha*], and Colorado pikeminnow [*Ptychocheilus lucius*]). However, unlike razorback suckers that continue to persist in reservoir situations, flannelmouth suckers apparently either did 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 suckers have been commonly captured in water temperatures ranging from 10 to 35 degrees Celsius (°C) (mean of 24 °C, conductivities varied from 150 to 2,700 micromhos) (Cross 1975). Deacon et al. (1987) found the final thermal preferences of flannelmouth suckers to be 25.9 °C ( ± 0.5 °C) using laboratory techniques, but they suggest that the upper temperature threshold determining habitat usage is highly dependent upon acclimation temperature and that flannelmouth suckers, in particular, have one of the highest temperature tolerances displayed by native fishes in 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 suckers. The 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 suckers and the potential impacts that cold-water releases may have on populations of this species. Based on studies conducted in southwestern Wyoming, Carter and Hubert (1995) found the upper elevational threshold for flannelmouth sucker inhabitance to be 2192 meters.

## Spawning

Flannelmouth suckers 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, spawning is known to occur in April and May in the LCRB (Mueller and Marsh 2002; Best and Lantow 2012). Historically, flannelmouth suckers, as well as other native catostomids, were reported to make impressive spawning migrations (Simon 1946; LaRivers 1962). More recently, relatively little migrational movement has been observed presumably due to the impacts associated with main stem impoundments (Holden 1973; McAda 1977; Best and Lantow 2012). McAda (1977) captured ripe male and female razorback suckers 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 suckers were visually observed and sampled below Davis Dam to Lake Havasu. Spawning fish were found congregating in water approximately 1–2 meters deep, over large cobble and gravel substrates, and in relatively swift currents (0.5–1.0 meter per second) (Mueller and Wydoski 2004; Best and Lantow 2012). To the best of our knowledge, flannelmouth suckers 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 suckers is also important and heavily influenced by flow regimes (Robinson et al. 1998a, 1998b). However, unlike razorback sucker larvae, young flannelmouth suckers apparently do not exhibit diel drift periodicity. They do appear to be associated with near-shore (defined as shoreline habitats with velocities < 0.2 meter per second), slackwater rearing habitat types, indicating at least some degree of active affinity for drift habits (Robinson et al. 1998a, 1998b). Larval flannelmouth suckers are found in shoreline backwaters, embayments, and other low-velocity slackwater habitats in the San Juan River and LCRB using larval seines (Brandenburg et al. 2005; Best and Lantow 2012).

## Juveniles

Juvenile flannelmouth suckers move out of backwaters and embayments 2–3 months after hatching (Best and Lantow 2012). This is generally indicated by their sudden disappearance from seine collections, which concentrate on these low-velocity habitats. Juveniles tend to move into main channel habitats with more velocity such as runs and edges of riffles (Holden 1999; Walters et al. 2012). This is very similar to habitat shifts that occur with roundtail (*Gila robusta*) and humpback chubs (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 suckers in the San Juan River, but they do not compare the use of those habitats with the 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 suckers mature between 4 and 6 years of age in the UCRB, or at lengths ranging from 391 to 421 millimeters (mm) (McAda 1977; McAda and Wydoski 1985). Fecundity of flannelmouth suckers is highly dependent on size and is therefore highly variable within age groups (see the "Age and Growth" section below for reasoning); an individual female flannelmouth sucker produces anywhere from 3,000 to 30,000 eggs. 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 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

The diet composition of flannelmouth suckers 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) describes 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 suckers 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]) documents seasonal and size-specific shifts in flannelmouth sucker food habits. For example, flannelmouth suckers less than 80 mm in length feed exclusively on copepods, while fish greater than 200 mm in length do not consume copepods. As a flannelmouth sucker increases 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 suckers, particularly in the later spring and summer months. Furthermore, Ephemeroptera nymphs increase in importance during late summer into fall. Overall, the food habits of flannelmouth suckers are highly dependent 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

Several estimates of natural mortality rates for flannelmouth suckers in the Grand Canyon are summarized in Walters et al. (2012) and range from 0.16 to 0.25; these estimates decline with age as the fish grow in size. Published growth estimates for flannelmouth suckers vary, and available information is highly dependent on age and habitat occupied; for details, see age and growth tables compiled by McAda (1977), Sweet et al. (2009), Best and Lantow (2012), and Walters et al. (2012). Robinson and Childs (2001) found juvenile growth rates to be highly and significantly correlated with water temperature and estimated that zero growth occurs at temperatures of 10.8 °C. Aside from length, numerous structures have been used to estimate age. McAda (1977) uses scales as a method for age determination and for back-calculation of growth of UCRB flannelmouth suckers. Annuli formation was found to occur in June and July, with scales forming on young flannelmouth suckers 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 among age groups. Overall, age-1 fish were found to be between 57 and 80 mm, age-2 between 128 and 167 mm, age-3 between 245 and 286 mm, age-4 between 351 and 370 mm, age-5 between 412 and 432 mm, age-6 between 440 and 475 mm, age-7 between 446 and 493, and age-8 between 456 and 514 mm (McAda 1977). McKinney et al. (1999) report growth for flannelmouth suckers collected below Lee’s Ferry, with fish displaying nearly isometric growth. Adults typically grew 5.5 mm per year (standard deviation 1.9 mm) and subadults 45.9 mm per 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 discernible age information compared with otoliths and smaller fin ray sections. Non-lethal aging techniques using the second pectoral fin ray have been developed for razorback suckers (Albrecht et al. 2008), and this method has been used to age populations of flannelmouth suckers in the LCRB (Best and Lantow 2012; Keggeries and Albrecht 2013). Length distribution data compiled by Mueller and Wydoski (2004) suggest that recruitment of flannelmouth suckers to the spawning cohort documented in the LCRB ranges between 9.4 and 31.3% per year, suggesting the capability of flannelmouth suckers to maintain a population of spawning-aged fish despite extensive riverine habitat modifications.

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# Flat-tailed Horned Lizard (*Phrynosoma mcallii*)

## LEGAL STATUS

The U.S. Fish and Wildlife Service (USFWS) published a proposed rule to list flat-tailed horned lizards (*Phrynosoma mcallii*) as a threatened species on November 29, 1993. No critical habitat was determined at that time (USFWS 2005). The proposal to list flat-tailed horned lizards was withdrawn on July 15, 1997, for three primary reasons: (1) population trend data did not conclusively demonstrate significant population declines, (2) some threats to the occupied habitat of the species 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, 2006). The 1993 proposal to list flat-tailed horned lizards was reinstated on December 26, 2001, withdrawn on January 3, 2003, reinstated on December 7, 2005, and withdrawn on June 28, 2006 (USFWS 2005, 2006). The Bureau of Land Management (BLM) designated flat-tailed horned lizards as a sensitive species in 1980 (Wright 2002). The Mexican Government has designated flat-tailed horned lizards as a threatened species (Flat-tailed Horned Lizard Interagency Coordinating Committee [FTHL ICC] 2003).

Flat-tailed horned lizards are listed as a species of special concern in Arizona and a species of concern in California (Arizona Game and Fish Department [AGFD] 2003). The California Fish and Game Commission designated flat-tailed horned lizards as a candidate species and recommended it be listed as a threatened species in California on May 13, 1988 (Bolster and Nicol 1989). The commission voted against the proposed listing on June 22, 1989 (Foreman 1997). Collecting flat-tailed horned lizards is prohibited in California and Arizona (AGFD 2003). NatureServe ranks flat-tailed horned lizards 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 flat-tailed horned lizards throughout the species' range in the United States (Foreman 1997). The document was revised in 2003 (FTHL ICC 2003).

# DISTRIBUTION

## Historical Range

Assessing the historical habitat of flat-tailed horned lizards was complicated by the fact that agricultural conversion occurred before the flat-tailed horned lizard's range in Imperial Valley, California, and Yuma Valley, Arizona, was well documented; therefore, it is unknown what portion of the agricultural habitat was historically used by the species (Hodges 1997). Hodges (1995) estimates that there are 203,520 acres of flat-tailed horned lizard historical habitat in Arizona. Piest and Knowles (2002) estimate that there are 221,043 acres of flat-tailed horned lizard historical habitat in Arizona. The northern and western boundary of their historical range in Arizona is the Yuma Mesa (figure 1) (Hodges 1997). The eastern boundary is formed by the Fortuna Wash and Foothills Boulevards to just east of the sand dunes (figure 1) (Hodges 1997). Hodges (1997) estimates that there were 2.22 million acres of flat-tailed horned lizard historical habitat in California. The western boundary of their historical range in California is formed by the 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 Interstate 8 beyond Ocotillo and Coyote Wells, where the interstate joins Highway 92 and S2 forks north (figure 1) (Hodges 1997). Borrego Valley, between the Vallecito and Santa Rosa Mountains, contains flat-tailed horned lizard habitat as does a valley between Indio Hills and the Little San Bernardino Mountains (figure 1) (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 and Cargo Muchacho Mountains (figure 1) (Hodges 1997). The existence of flat-tailed horned lizards in Carrizo Valley, which is south of the Fish Creek Mountains and north of the Coyote Mountains, is unknown (Hodges 1997). The western limit and distribution in Borrego Valley is still relatively unknown (figure 1) (Hodges 1997).

## Current Range

Flat-tailed horned lizards occur 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 (figure 1) (AGFD 2003; California Department of Fish and Game [CDFG] 1994; Rodrigues 2002). Flat-tailed horned lizards have the smallest range of any of the horned lizards in the United States (Wright 2002).

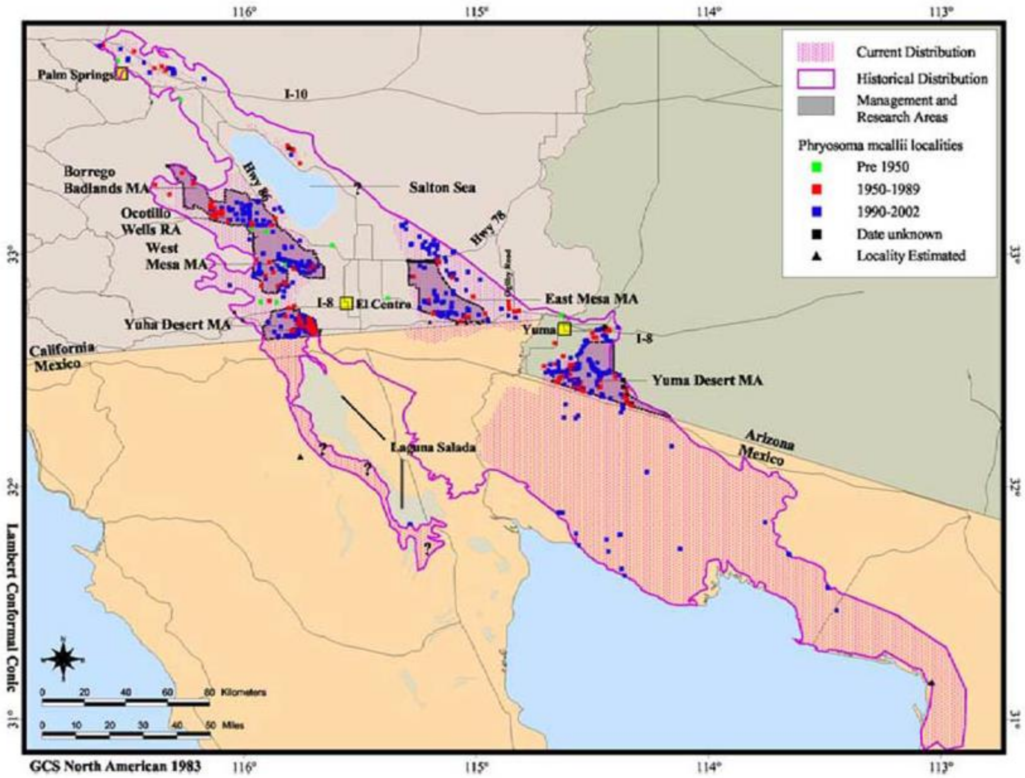

**Figure 1.—The flat-tailed horned lizard's range in the United States and Mexico.**

The flat-tailed horned lizard's range in California extends from near the confluence of the San Gorgonio and Whitewater Rivers in Riverside County, south and east through the Coachella Valley into Imperial County along both sides of the Salton Sea (see figure 1 ) (Wright 2002). From the area between the Salton Sea and the 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 (see figure 1) (Wright 2002). Flat-tailed horned lizards are generally restricted to elevations below 250 meters 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 (see figure 1) (Wright 2002). Flat-tailed horned lizards occur 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 (see figure 1) (Wright 2002). Flat-tailed horned lizards occur east of Bow Willow in the Carrizo Wash (see figure 1) (Wright 2002). They may occur in other flats or bajadas along the Carrizo Wash and in drainages within the Carrizo Badlands. The species' 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 (see figure 1) (Wright

2002). 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). The flat-tailed horned lizard's range extends from the Borrego Badlands to Pilot Knob Mesa, east of the Algodones Dunes (see figure 1) (Wright 2002). They occur throughout much of the sandy flats and dune margins in the southern portion of the Salton Sea Test Base (Rorabaugh 1996a, 1996b) (see figure 1). Flat-tailed horned lizards inhabit 2,695 square miles in California; 330 square miles are considered optimal habitat (Rado 1981).

The flat-tailed horned lizard's range in southwestern Arizona extends southward of the Gila River and west of the Gila and Tinajas Atlas Mountains in Yuma County (see figure 1) (Townships T.9S, T.10S, T.11S, and T.12S; Ranges R.24W, R.23W, R.22W, R.21W, and R.20W) (NatureServe 2005; AGFD 2003; Rorabaugh et al. 1987). Hodges (1995) estimates that the species inhabits 550–575 square kilometers in Arizona. Rorabaugh et al. (1987) estimate that flat-tailed horned lizards inhabit 650–700 square kilometers in Arizona. Their 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 (see figure 1) (NatureServe 2005; Rodrigues 2002). Flat-tailed horned lizards do not occur contiguously across their range.

There are four geographically discrete populations of flat-tailed horned lizards 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). The range of flat-tailed horned lizards and desert horned lizards (*Phrynosoma platyrhinos*) overlap in portions of Arizona (Township T12S, Range 20/21W) and California (Hodges 1995; Rorabaugh et al. 1987).

Additional surveys are still needed outside the flat-tailed horned lizard management areas (Yuma Desert, East Mesa, West Mesa, Yuha Basin, and the Borrego Badlands) to more accurately delineate the boundaries of the flat-tailed horned lizard's range in the United States (Foreman 1997). Distribution of the species in Mexico is poorly understood because few surveys in Mexico have been conducted (NatureServe 2005), especially in the southeast boundary in Sonora, the current range surrounding Mexicali Valley, and the current range surrounding the Laguna Salada. The degree to which current populations interact with each other in Sonora, the Mexicali Valley, and surrounding Laguna Salada is also poorly understood (Foreman 1997).

## Populations Within the LCR MSCP Planning Area

Flat-tailed horned lizards occur within Reaches 6 and 7 of the Lower Colorado River Multi-Species Conservation Plan (LCR MSCP) planning area (see figure 1) (Bureau of Reclamation [Reclamation] 2004). The Yuma Desert Management Area is the only flat-tailed horned lizard management area that lies within the LCR MSCP planning area (see figure 1)(Foreman 1997).

## Management and Research Areas

Five management areas and one research area were established for flat-tailed horned lizards in the 1997 Rangewide Management Strategy (Foreman 1997). Management areas include the majority of the flat-tailed horned lizard's 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 landowners (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) (see figure 1) (Foreman 1997). A research area at Ocotillo Wells State Vehicle Recreation Area (OWSVRA), encompassing 76,700 acres, is used for research on the flat-tailed horned lizard (see figure 1). Research priorities focus on the effects of off-highway vehicle (OHV) use on flat-tailed horned lizard 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

The flat-tailed horned lizard's relative abundance indices in Arizona are greatest (> 15) in the four townships that are in the south-central portion of the species' range (Rorabaugh et al. 1987). In California, the species 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) uses closed mark-recapture analyses to estimate population sizes at three management areas from 2002 to 2004. Population estimates for the Yuha Basin Management Area in 2002 and 2004 were 25,514 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 and 10,849 lizards, respectively (Grant 2005). No overall trend could be inferred in the Yuha Basin Management Area from 2002 to 2004, as confidence intervals overlapped (Grant 2005). The population estimate for the Yuma Desert Management Area using a mark-recapture analysis was 25,855 lizards (FTHL ICC 2003). The population estimate for the OWSVRA was 19,222 lizards (FTHL ICC 2003).

# LIFE HISTORY

## General Description

One of 14 species in the genus *Phrynosoma,* flat-tailed horned lizards were first collected by Colonel George A. McCall and described by Hallowell in 1852 (Funk 1981; AGFD 2003). No subspecies of flat-tailed horned lizards has been described (Funk 1981). Flat-tailed horned lizards are 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 three to four 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 millimeters (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 grams (Bolster and Nicol 1989; Young and Young 2000). Gardner and Foley (2001) found SVLs to be significantly correlated with weight.

Desert horned lizards are the only other horned lizards known to be sympatric with flat-tailed horned lizards. 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

Flat-tailed horned lizards mate in April and May, and first clutches are laid in May and June (AGFD 2003). Howard (1974) found that male flat-tailed horned lizards 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 flat-tailed horned lizard eggs were present in May, and hatchlings were present from July to October. They lay approximately 3 to 10 eggs per clutch and have up to 2 clutches per season (CDFG 1994; Muth and Fisher 1992). The average clutch size is 4.7, and there is a significant correlation between body size and clutch size of this species (Howard 1974). Flat-tailed horned lizards follow the reproduction strategy of multiple small clutches and early reproduction (Howard 1974). Howard (1974) found that of seven lizards in the *Phrynosoma* genera, flat-tailed horned lizards had the lowest productivity index. They reach sexual maturity at age-1 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). The sex ratio of flat-tailed horned lizards is 1:1 (male: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 and Doherty 2007). 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; 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 flat-tailed horned lizards (Young and Young 2000). They are obligate hibernators that overwinter at 2.5 to 20 centimeters of depth in loose sand (CDFG 1994). The average depth of hibernation burrows in one population of flat-tailed horned lizards was 6.0 centimeters (Grant 2005). The entrance of hibernation burrows is plugged with substrate (Grant 2005). The flat-tailed horned lizard’s hibernation behavior is variable. Grant (2005) observed them entering hibernation burrows from early October to late December. Some individuals abandon their first hibernation burrows and dig new ones (Grant and Doherty 2007). Body mass affects the date that lizards enter into hibernation. Flat-tailed horned lizards delay hibernation to continue to grow or gain mass reserves for hibernation (Grant and Doherty 2007). Several studies have estimated average winter dormancy of the species at 85–89 days (Wone and Beauchamp 2003; Muth and Fisher 1992). Winter dormancy of flat-tailed horned lizards is controlled primarily by a reduced photoperiod and reduced air temperature rather than a reduced metabolic rate or body temperature (Mayhew 1965). Flat-tailed horned lizards emerge from overwintering sites from late December through March when substrate temperatures at a depth of 5 centimeters reach their voluntary minimum temperature (CDFG 1994; Wone and Beauchamp 2003). Some juveniles remain active during the winter months (Grant and Doherty 2007; NatureServe 2005).

Miller (1999) found that the area used by male and female flat-tailed horned lizards during 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 hectares (ha), and females used an average of 1.77 ha during 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 use significantly more area than males do (Miller 1999). Turner and Medica (1982) estimate the home range size for flat-tailed horned lizard males and females to be 0.12 and 0.05 ha, respectively. Muth and Fisher (1992) estimate the home range size for flat-tailed horned lizard males and females to be 1.78 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). Flat-tailed horned lizards show considerable overlap between home ranges, which may indicate lack of territorial behavior (Wone and Beauchamp 2003). Their home range is significantly larger during breeding season than during their non-breeding season (Wone and Beauchamp 2003).

Flat-tailed horned lizards reach their peak abundance from the months of April through October (Muth and Fisher 1992). Rainfall appears to be a factor in their abundance (Wright 1993, 2002; Wright and Grant 2003; Grant 2005). Flat-tailed horned lizard 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 flat-tailed horned lizards spent 32% of the active period moving, 46% of the time they were motionless, 11% of the time they were feeding, and 11% of the time they were digging. They spend the night on the surface, fully exposed in the open (Young and Young 2000). Flat-tailed horned lizards appear to partake in sand swimming for short distances, but swimming does not occur after the loss of momentum. They are capable of rapid locomotion, but this is not sustained for long distances (Norris 1949). Flat-tailed horned lizards avoid predators by diving in the sand in areas where Aeolian sand is present. When sand is absent, they 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, they wiggle their head and dig their occipital horns into the handler's hand, which may be an important escape behavior when captured by predators (Wone and Beauchamp 1995).

The optimal air temperature for flat-tailed horned lizards appears to be 35.2 to 40.2 degrees Celsius (°C); when temperatures exceed 41 °C, individuals retreat underground (CDFG 1994; Wone and Beauchamp 2003; Wright 2002). They maintain an optimal body temperature by orienting their body toward the sun or substrate (Norris 1949). The foraging strategy of flat-tailed horned lizards is intermediate to that of a sit-and-wait predator and active forager (Muth and Fisher 1992).

## Diet

Flat-tailed horned lizards 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 flat-tailed horned lizard 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 flat-tailed horned lizards. Results of several studies indicate that the number of harvester ant colonies is correlated with high lizard abundance (Grant 2005; Rorabaugh et al. 1987; Turner and Medica 1982; Young 1999).

Flat-tailed horned lizards primarily use pre-formed water (water found in their food) to maintain proper water balance (AGFD 2003). Rain harvesting may provide an important source of water for the species. Grant (2005) observed the first known occurrence of rain harvesting in flat-tailed horned lizards 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 increases after a rainfall event, which is more evidence that this species uses rain harvesting as a means of ingesting water (Grant 2005). Flat-tailed horned lizards are not capable of using their skin to collect water from the environment (Mayhew and Wright 1971).

## Habitat

Flat-tailed horned lizards occur 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). They occur at elevations from below sea level to 250 meters (AGFD 2005). Vegetation may be an important requirement for oviposition sites (CDFG 1994). Flat-tailed horned lizards do not normally occur in a number of habitats represented within their 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 salt cedar-arrowweed thickets, and (6) agricultural and developed areas (Turner et al. 1980).

There have been several studies conducted correlating flat-tailed horned lizard relative abundance to habitat characteristics. Results of several studies indicated that high flat-tailed horned lizard relative abundance correlated with the percent of sandy substrate (Grant and Doherty 2007; Hodges 1995; Muth and Fisher 1992; Rorabaugh et al. 1987). Wright (2002) found no significant difference between flat-tailed horned lizard relative abundance and substrate type (sand, gravel, or hardpan). Grant and Doherty (2007) believe that flat-tailed horned lizards may prefer sandy areas but are not necessarily confined to sandy areas as once believed. Gardner and Foley (2001) observed that flat-tailed horned lizards used two different types of substrate: (1) compacted sand with a shallow surface of loose-grained sands and (2) loose, small- to medium-grain sand.

Wone and Beauchamp (1995) observed flat-tailed horned lizards in hardpan soil covered with gravel and sparse vegetation. Turner and Medica (1982) found that high relative abundance positively correlated with perennial density and diversity. Barrows and Allen (2009) found that flat-tailed horned lizards are associated with fine, moderately active Aeolian sands. Grant (2005) found no correlation between perennial density and flat-tailed horned lizard relative abundance. Hodges (1995) found that plant density, diversity, and percent cover were not correlated with their relative abundance. Rorabaugh et al. (1987) found high flat-tailed horned lizard relative abundance in areas where galleta grass dominated. Muth and Fisher (1992) found that flat-tailed horned lizards preferred white bursage and indigo bush but avoided creosote bush and coldenia (*Tequilia plicata*).

Beauchamp et al. (1998) found that high flat-tailed horned lizard abundance was correlated with large patches of concretions, gravel, silt, and sparse perennial vegetation at the OWSVRA in California. High relative abundance was negatively correlated with dense perennial vegetation (Beauchamp et al. 1998). Beauchamp et al. (1998) also found flat-tailed horned lizards using mudhills, which suggests that either flat-tailed horned lizards have shifted or dispersed to other habitats because of OHV use in sandy areas or that the species has a wider habitat preference than previously described (Beauchamp et al. 1998).

The above studies have all found flat-tailed horned lizard relative abundance to be correlated with some habitat characteristic; many of these characteristics differ among 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 flat-tailed horned lizards, there is a lack of data on which characteristic is the most important in determining their relative abundance. More research is needed to determine the most important habitat requirement for this species (Rorabaugh et al. 1987; Turner and Medica 1982). It has always been thought that flat-tailed horned lizards were primarily associated with sandy areas, but they have been observed in other places. Whether this is due to a disturbance of sandy habitat, or whether flat-tailed horned lizards have wider habitat preferences than previously thought, is not conclusive. The types of habitat used by flat-tailed horned lizards across their range needs to be re-evaluated (Beauchamp et al. 1998).

## Threats

Historically, flat-tailed horned lizard habitat loss occurred due to the creation of the Salton Sea, agricultural conversion, and human expansion. Current threats to the species include habitat loss from urban and agricultural expansion, pesticide contamination, OHV 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). Agricultural expansion and urban development have an indirect effect on adjacent flat-tailed horned lizard populations up to 450 meters 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 flat-tailed horned lizard habitat and act as barriers to movement (FTHL ICC 2003).

Hodges (1997) estimates that 63,129 acres (31.10%) of historical flat-tailed horned lizard 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) estimates that 1,112,640 acres (50.20%) of historical flat-tailed horned lizard habitat in California has been lost due to flooding of the Salton Sea, agricultural conversion, urban development, military activities, and fire. Hodges estimates 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 flat-tailed horned lizards 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 (Reclamation 1996). Activities in this area include the operation and maintenance of the 242 Well Field, operation of the Yuma Desalting Plant sludge disposal site, and maintenance of the canal used for delivery of water to Mexico (Reclamation 1996). There are approximately 40 records of flat-tailed horned lizards within this zone (Reclamation 1996). Reclamation has preserved 16,000 acres in the Yuma Desert Management Area (Reclamation 1996). Future threats to flat-tailed horned lizards 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 (Reclamation 2004).

Vehicular traffic is a direct threat to flat-tailed horned lizard populations; many individuals are killed by vehicles when on the road. The primary defense behavior of the species 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 flat-tailed horned lizards within at least 500 meters of either side of the road, with severe impacts within 250 meters of the road (Young and Young 2000). Every kilometer of road would potentially impact 100 ha of habitat.

Gardner et al. (2004) designed fencing that was successful in keeping flat-tailed horned lizard off roads. The fence was constructed with four rolls (30.5 meters long, 90 centimeters high) of hardware cloth, with 6.5-mm (1/4-inch) mesh, buried to a depth of 15 centimeters and attached with cable ties to rebar supports at 2.5-meter intervals. Fences were located 5 meters from the edges of roads (Gardner et al. 2004). Suggested improvements to the design included 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-sized culverts and skylights (Painter and Ingraldi 2005). Other strategies to reduce fatalities by vehicles are to allow vehicle traffic on roads only during the flat-tailed horned lizard’s 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 and 35 °C (Young and Young 2000).

OHV usage is an increasingly popular activity that takes place in flat-tailed horned lizard habitat. OHV usage may pose direct threats (mortality by being run over) or indirect threats (destroying ant mounds, affecting vegetation, compacting soil, and damage to hearing) to flat-tailed horned lizard populations (Grant and Doherty 2009). Studies on impacts of OHV use on the species are incomplete and inconclusive (FTHL ICC 2003). Wright (1993) found that flat-tailed horned lizard 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 flat-tailed horned lizard detection rates in the Yuha Basin. Grant and Doherty (2009) found that OHV activity did not directly affect flat-tailed horned lizard populations during hibernation, which is the main OHV season. Setser (2004) did not observe any direct flat-tailed horned lizard mortality due to OHV activity. Rates and direction of flat-tailed horned lizard movement differed significantly before OHV races versus after OHV races, but the impact that those findings have on the species is unknown (Nicola and Lovich 2000). Flat-tailed horned lizards may have shifted habitat use from sandy areas to other areas (concrete, mudhills, gravel, or silt) at the OWSVRA due to OHV use (Beauchamp et al. 1998). More research on the direct and indirect effects of OHV activity on the species is needed.

The Border Patrol conducts patrols and rescues near the international border that sometimes involve cross-country travel. Border Patrol activities in flat-tailed horned lizard 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, 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 populations 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 flat-tailed horned lizards include round-tailed ground squirrels (*Spermophilus tereticaudus*) and loggerhead shrikes (*Lanius ludovicianus*). Other predators of the species include grasshopper mice (*Onychomys* spp.), American kestrels (*Falco sparverius*), common ravens (*Corvus corax*), burrowing owls (*Athene cunicularia*), snakes, and feral cats and canids (AGFD 2003; Duncan et al. 1994; NatureServe 2005).

## Flat-tailed Horned Lizard Rangewide Management Strategy

A rangewide management strategy for flat-tailed horned lizards 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 the 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, 2006).

## Survey Methods

A variety of methods have been used to estimate flat-tailed horned lizard relative abundance throughout their range. Surveys conducted by Beauchamp et al. (1998) consisted of belt transects spaced 20 meters apart that were surveyed four times between June and July. Observers counted all the flat-tailed horned lizards and their scat that was greater than 5.5 mm in diameter (Beauchamp et al. 1998). Surveys conducted by Hodges (1995) consisted of belt transects spaced 5 meters apart that were surveyed three times between April and August. Observers counted all flat-tailed horned lizards and their scat that was 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 flat-tailed horned lizard relative abundance. A section search is a 1-hour walk through a 2.50-square-kilometer area where observers count the flat-tailed horned lizards and their scat (Turner and Medica 1982). Turner and Medica (1982) suggest that relative abundance estimates should be based on several section searches per township. The BLM used 2.5-mile triangular transects where observers walked transects in 1 hour and counted all the lizards and scat greater than 5.5 mm (Olech 1986; Wright 1993, 2002). Wright (2002) suggests that at least 55 triangular transects should be conducted per area assessed for flat-tailed horned lizard 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, and high) between survey methods.

All of these methods relied upon systematic counts of scat as an accurate assessment of flat-tailed horned lizard abundance. The species produces large, visible scat that consist mostly of ant parts (Rorabaugh et al. 1987). Scat counts are an attractive alternative to direct enumeration of flat-tailed horned lizards 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 indices are a reliable indicator of flat-tailed horned lizard abundance and distribution. Annual differences in scat counts may be influenced by the rate of scat production per lizard rather than the abundance of the species (Rorabaugh 1994). Scat counts are affected by wind, heavy rains, and observer bias (Rorabaugh 1994). Scat may have a clumped distribution within the species' home range; it 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.

The results of several studies have indicated that flat-tailed horned lizard scat counts and the number of flat-tailed horned lizards were not correlated (Beauchamp et al. 1998, Hodges 1995). Wright (1993) found a significant correlation between flat-tailed horned lizards and their scat per hour, but a regression analysis showed that scat per hour is a poor predictor of the actual number of flat-tailed horned lizard sightings. Flat-tailed horned lizard scat is only distinguishable from desert horned lizard scat if it is greater than 5.5 mm in diameter (Muth and Fisher 1992). Rorabaugh (1994) suggests that scat counts should be used cautiously and combined with habitat evaluations and locality of records of flat-tailed horned lizards in assessing the importance of habitat areas for this species. Muth and Fisher (1992) recommended that scat counts 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 the flat-tailed horned lizard's range due to inconsistent monitoring protocols and the inaccuracy of scat counts (Foreman 1997). Mark-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-recapture monitoring protocol based on Wright and Grant (2003) and a standardized distribution monitoring protocol.

Mark-recapture methods have been used to assess flat-tailed horned lizard 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-meter plot sampled by twenty 400-meter-long belt transects spaced 20 meters apart produced statistically valid samples to be used in a mark-recapture study. Wone et al. (1994) made the following suggestions with regard to mark-recapture studies: (1) mark flat-tailed horned lizards by filing a notch on their occipital horn, (2) juvenile flat-tailed horned lizards 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 the population size of flat-tailed horned lizards.

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-recapture method included using a covariate to take observer bias into account and increasing the sampling fraction of all areas (Grant 2005). Grant (2005) suggests that further studies on distance sampling should have an emphasis on trying to estimate detection probabilities at various distances from the observer. Grant (2005) stresses the importance of sample size in mark-recapture methods and distance sampling methods. Occupancy estimation is another technique recently used to monitor flat-tailed horned lizard populations; it is less time consuming than mark-recapture, allowing for more area to be covered (BLM 2005). Another technique discussed in the literature is to combine habitat protection with presence/absence surveys for flat-tailed horned lizards (Young and Young 2000). Young and Young (2000) feel 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 this species occurs 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.

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# Gila Woodpecker (*Melanerpes uropygialis*)

## LEGAL STATUS

Gila woodpeckers (*Melanerpes uropygialis*) are not a federally listed species. California listed them as endangered in 1980 (California Department of Fish and Game 1980). Gila woodpeckers are not listed in either Arizona or Nevada.

## DISTRIBUTION

### Historical Range

There are no known fossil records of Gila woodpeckers. Historically, they were 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, Gila woodpecker populations have declined in southeastern California from their historical range. This decline is associated with the loss of woodland habitat and competition with European starlings (*Sturnus vulgaris*) (Garrett and Dunn 1981; Hunter 1984; Rosenberg et al. 1991; Kaufman 1996). Gila woodpeckers still occupy their historic range along the LCR, in extreme southern Nevada, southern Arizona, and throughout western Mexico. A few rare sightings outside of their range have been documented. Hubbard (1978) reported Gila woodpeckers in extreme southwestern New Mexico, Willett (1933) reports individuals in Los Angeles County, and Garrett and Dunn (1981) report individuals in Ontario, California. Figure 1 illustrates the Gila woodpecker's current range.

Grinnell (1914) found Gila woodpeckers "at every station on both sides of the river" during his trip down the Colorado River. Coues (1866) reports Gila woodpeckers as abundant along the LCR. More recently, Hunter (1984) reports 200 breeding individuals in California, with about one-half on private lands and in parks. Along the LCR, Gila woodpeckers can be found as far north as

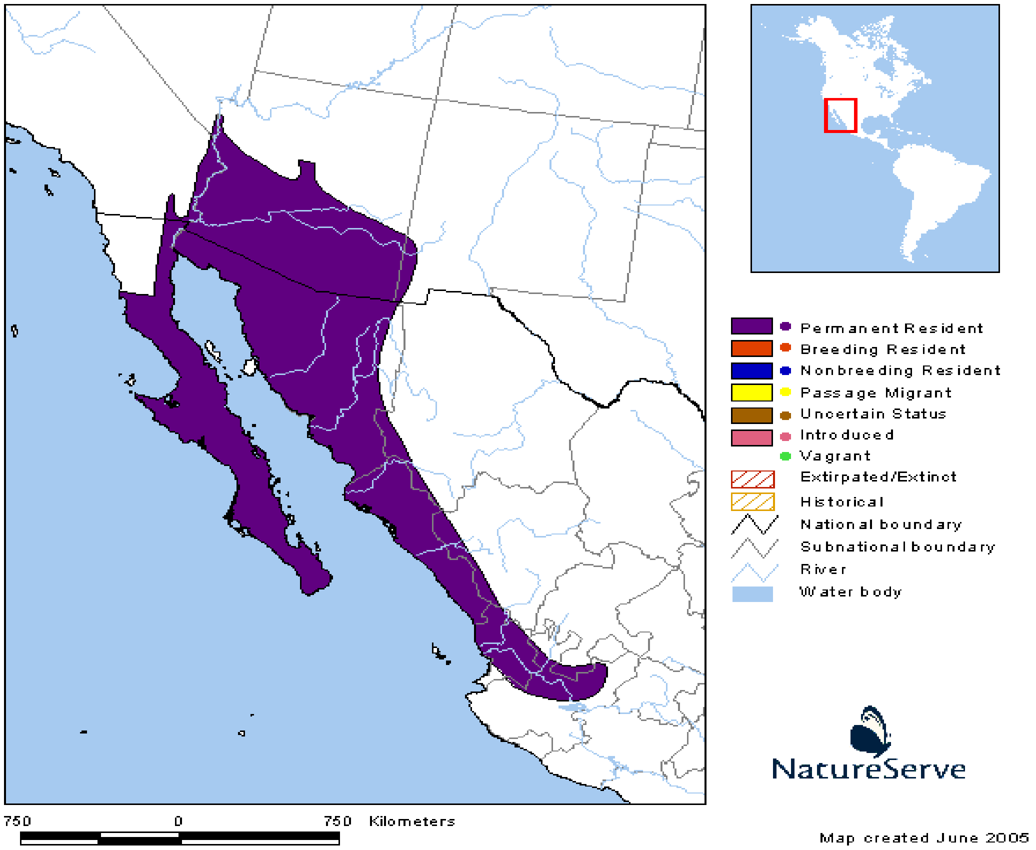

**Figure 1.—Populations of Gila woodpeckers within the Lower Colorado River Multi-Species Conservation Program project boundary.**

Clark County, Nevada, and south to Yuma, Arizona (Hollister 1908; Bureau of Reclamation [Reclamation] 2006a, 2006b). Rosenberg et al. (1991) suggest 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 1,000 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.

General bird surveys coordinated under the Lower Colorado Region Multi-Species Conservation Program (LCR MSCP) that are used to sample all riparian habitat within the LCR MSCP planning area and along the Bill Williams River have been taking place since 2007 (Great Basin Bird Observatory [GBBO] 2008a, 2008b, 2009, 2011, 2012, 2013). Approximately 400 to 1,000 pairs have been estimated to breed along the LCR and the Bill Williams River annually (GBBO 2008a, 2008b, 2009, 2011, 2012, 2013). Gila woodpeckers have not been detected in any of the LCR MSCP habitat conservation areas (GBBO 2008a, 2008b, 2009, 2011, 2012, 2013).

# LIFE HISTORY

## General Description

Gila woodpeckers are noisy, aggressive, and conspicuous medium-sized woodpeckers. Adults weigh 51–79 grams and measure 24 centimeters from head to tail. Adults have a light grayish-brown head, neck, and underparts. The bill is dull black. The abdomen is golden yellow. 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 a close distance, males have conspicuous red patches on the top of their heads. Females and juveniles resemble the male; however, they lack the red patch on their head. Dimorphic in size, the male is 14% heavier, with a 14% longer bill.

## Vocalizations

Categorized as loud and aggressive, Gila woodpeckers regularly produce two distinct calls. Bendire (1895) and Gilman (1915) describe call 1 as the "sociable" call and call 2 as 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). The duration of notes ranged from 0.18 to 0.95 second. 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 a 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 standard deviation, n = 29). The intervals between the notes were 0.08 to 0.14 second. 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 is shorter in duration and with an emphasis of different harmonics. This call was heard only when Gila woodpeckers were 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 describes it as an example of "ambivalent behavior," containing components of conflicting tendencies (self-advertisement and alarm).

## Migration

A non-migratory species, Gila woodpeckers may roam locally short distances in winter as food sources move (Kaufman 1996).

## Habitat

Found in the arid deserts of the Southwestern United States and northeastern Mexico, Gila woodpeckers use saguaro cacti (*Carnegiea gigantea*) and riparian woodlands (Gilman 1915; Bancroft 1929). Within the LCR MSCP planning area, they are found along the LCR, its tributaries, 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) in the Havasu National Wildlife Refuge (Reclamation 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 used habitats that Anderson and Ohmart (1984) defined as cottonwood-willow types I, II, and III.

A research study on preliminary habitat associations coordinated under the LCR MSCP showed that Gila woodpeckers tend to be associated with a high canopy, large trees and snags, and the presence of willow trees (GBBO 2011). Under the LCR MSCP, a research study is currently being coordinated that will help provide for the creation of more indepth habitat associations for this species.

## Diet

The main food sources of Gila woodpeckers are insects such as ants, beetles, grasshoppers, and cicadas as well as moths and butterflies. The fruits of 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, they 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 planning 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 (Isoptera), beetles, insect larvae, and a few cactus fruits in their stomachs.

## Behavior

The flight of a Gila woodpecker 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) describe Gila woodpeckers as highly aggressive toward both con- and heterospecifics. In Brenowitz (1978b), they exhibited agnostic behavior toward other Gila woodpeckers, common flickers (*Colaptes auritus*), and European starlings during breeding. The agnostic behaviors included supplanting, chasing, and attacking with their bill. The most aggressive behaviors were related to defense of their 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 hectares (Hensley 1954). Gila woodpeckers in washes had smaller territories, while larger territories were established in 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 lower Colorado River 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 their cavities (Gilman 1915; Brush 1983). Most breeding data come 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). Korol and Hutto (1984) report Gila woodpeckers disproportionately using taller saguaros with non-random oriented cavity entrances. Inouye et al. (1981) also report non-random-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) also note that Inouye et al. (1981) and Korol 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 are available for age at first breeding or 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) estimates the total population of Gila woodpeckers to be 200 individuals from surveys conducted during 1975–79.

Rosenberg et al. (1991) estimates 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 provide an estimated population trend decline ranging from -1.5 to -3.4% from 1966 to 2004. GBBO (2008a, 2008b, 2009, 2011, 2012, 2013) estimated that 400 to 1,000 Gila woodpecker pairs were breeding within the LCR MSCP planning area and along the Bill Williams River (confluence of Lake Havasu to Alamo Dam) annually from 2007 to 2012.

## 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 woodpeckers using nest boxes or artificial snags. Brush (1983) has the only data on cavity dimensions, recording mean cavity entrance diameters of 5.4 centimeters (n = 7) and cavity depths of 26.4 centimeters (n = 5). Mean cavity heights of 1.5 meters (n = 6) in mesquite and 10.3 meters (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) reports 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 hectares in area were devoid of Gila woodpeckers. Tweit and Tweit (1986) show that urban residential housing development, at a density of 2 houses per hectare, does not reduce the number of Gila woodpeckers, provided native vegetation is maintained.

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# Gilded Flicker
# (*Colaptes Chrysoides*)

## INTRODUCTION

A review of the literature indicates that very little research has been conducted on gilded flickers (*Colaptes chrysoides*). Most of the available research has been conducted on northern flickers (*Colaptes auratus*) and the subspecies of red-shafted flickers (*C. auratus cafer*) and yellow-shafted flickers (*C. a. auratus*). The American Ornithologists' Union has considered gilded flickers a subspecies of, or the same species as, northern flickers several times in the past. Therefore, past research on northern flickers in the Southwest may have actually been conducted on gilded flickers; however, this cannot be substantiated. In this document, the term flicker will refer to the *Colaptes* genus.

## LEGAL STATUS

Gilded flickers are not a federally listed species. California listed them as endangered in 1988 (California Department of Fish and Game 1988). Gilded flickers are not listed in either Arizona or Nevada.

## DISTRIBUTION

### Historical Range

The historical range of gilded flickers has not been adequately described. Taxonomic differences within the genus *Colaptes* have been confounded over the past 100 years. Fossil records indicate that gilded flickers 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, they are from the genus *Colaptes* and not necessary *C. chrysoides*.

### Current Range

Gilded flickers breed mostly where saguaro (*Carnegiea gigantea*) and Joshua trees (*Yucca brevifolia*) exist along the lower Colorado River (LCR), in southern Nevada, southern Arizona, and eastern California, south to the tip of Baja

California, Mexico, and west through the Sonoran Desert (NatureServe 2013). Typically, gilded flickers are absent in urban areas within their range. Figure 1 is a map of the gilded flicker's distribution as described above (Ridgely et al. 2003).

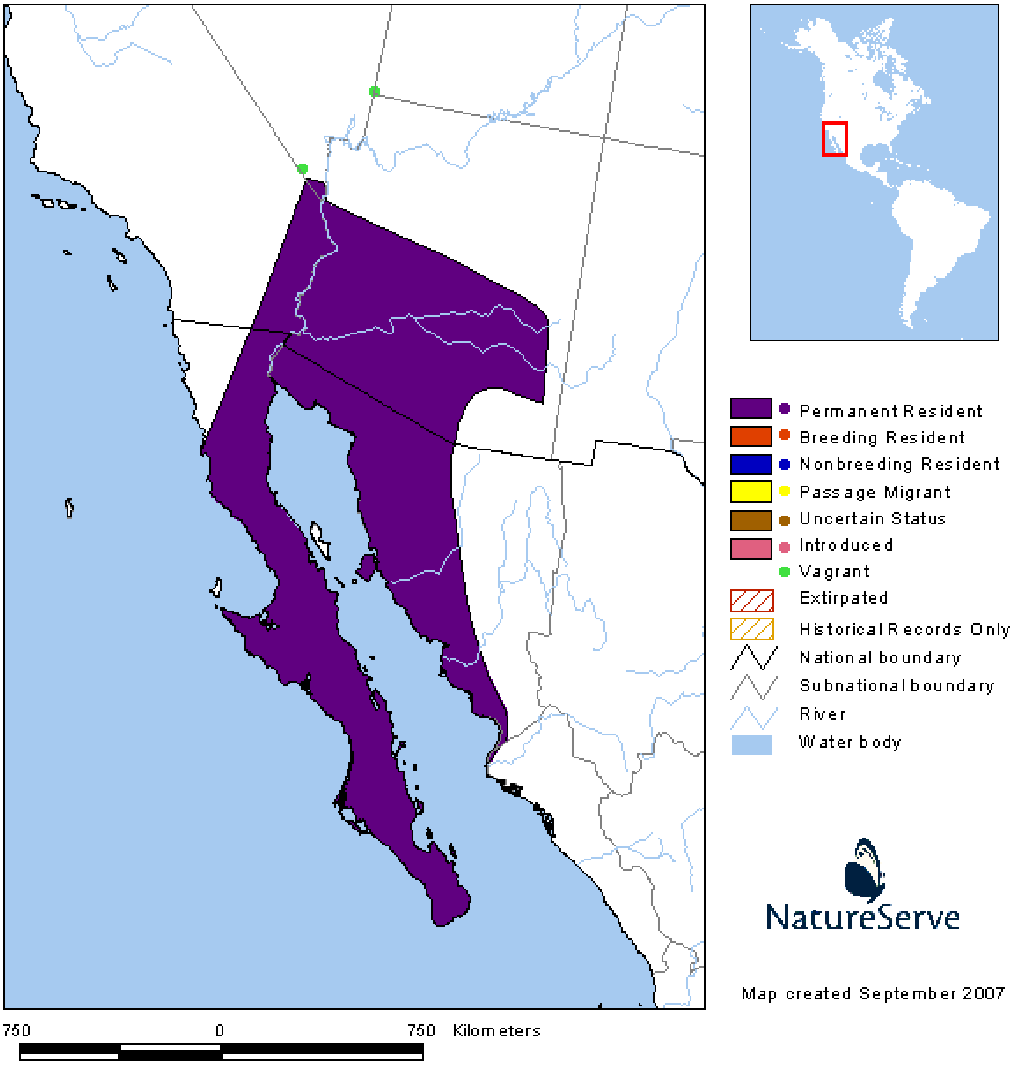

**Figure 1.—Gilded flicker distribution.**

## Populations Within the LCR MSCP Planning Area

At Fort Mohave, Arizona, Cooper (1870 *in* Grinnell 1914) found two pairs of gilded flickers nesting in cottonwood (*Populus* spp.) trees. Brown (1904) found a pair north of Laguna Dam nesting in a saguaro cactus. Grinnell (1914) found two pairs nesting in dead cottonwood stumps just north of Laguna Dam in the Potholes region. Grinnell (1914) also found three pairs nesting in the saguaro belt north of Laguna Dam and noted that the birds were fairly common in this area. Grinnell (1914) notes many inactive cavities in cottonwood and willow (*Salix* spp.) trees from Pilot Knob (in California, just west of Yuma, Arizona) north to above the Picacho State Recreation Area. The observed inactive cavities, found in cottonwood, willows, and saguaros, were probably cavities excavated for nest cavities in previous years and attributed to gilded flickers, as they were too large for Gila woodpeckers (*Melanerpes uropygialis*) (Grinnell 1914). Swarth (1914) records the status of gilded flickers along the Colorado River occurring at the few points where saguaro cacti are found. 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 in the 1970s and 1980s. Scattered pairs were detected breeding in the 1970s and 1980s along the main stem of the LCR at the following locations: Fort Mohave, the Colorado River Indian Reservation, Cibola and Imperial National Wildlife Refuges, Clark Ranch, near Blythe on the California side of the river, the Imperial Irrigation District, and between Imperial and Laguna Dams (Hunter 1984; Serena 1981; Rosenberg et al. 1991). Gilded flickers have been detected along the main stem of the LCR and at the Bill Williams River Delta from 1998 to 2012 during system-wide surveys for yellow-billed cuckoos (*Coccyzus americanus occidentalis),* southwestern willow flycatchers (*Empidonax traillii extimus*), and during general bird surveys (McKernan and Braden 2002; Halterman et al. 2009; McLeod and Koronkiewicz 2009, 2010; McNeil et al. 2010; McLeod and Pellegrini 2011; McNeil et al. 2012; Great Basin Bird Observatory [GBBO] 2011, 2012). The only location where breeding was confirmed during these surveys was in the saguaro habitat near Lincoln Ranch (GBBO 2011, 2012). One pair was observed foraging in the riparian habitat but nesting in the saguaros near Lincoln Ranch (GBBO 2011, 2012).

The Arizona Breeding Bird Atlas records gilded flickers in the Cibola and Imperial National Wildlife Refuges; however, breeding was not confirmed (Corman 2005). There was a probable breeding pair at McIntyre Park near Blythe, California, in 2012. The nest cavity was never located, so breeding could never be confirmed, but there were several cavities in the mulberry trees at the park and several saguaros across the river from the park on the Arizona side. The pair was observed in both places (B. Sabin 2012, personal communication).

Incidental sightings of gilded flickers have been noted during other standardized surveys such as the Breeding Bird Survey (BBS), the Christmas Bird Count, and U.S. Fish and Wildlife (USFWS) refuge surveys (Neale et al.1994; Lynn and Averill 1996; National Audubon Society 2002; K. Blair 2012, unpublished data; USGS Patuxent Wildlife Research Center 2012). Incidental sightings of gilded flickers have also been noted outside of standardized surveys from various biologists and other birders (B. Sabin 2013, unpublished data; M.E. Given 2013, unpublished data; eBird 2013). The only breeding confirmations along the LCR corridor have been on the Bill Williams National Wildlife Refuge near the Mohave and Mineral Washes (Neale et al. 1994; eBird 2013).

## Surveys and Population Estimates

Root (1988) utilizes national Christmas Bird Count data from 1963–72 to determine winter abundance of flickers. Gilded flicker abundance was recorded at 1.6 per hour (individual birds observed per hour), less than the 4.72 per hour recorded for red-shafted flickers and the 2.77 per hour recorded for yellow-shafted flickers. BBS data from 1966 to 2005 shows national gilded flicker population trends decreased 0.85% (Sauer et al. 2005). The majority of the BBS routes that have gilded flicker data occur outside of the LCR, in southern Arizona.

It appears that gilded flickers were apparently uncommon residents throughout the lower Colorado River Valley, being associated primarily with saguaro cacti and secondarily with cottonwood forests (Grinnell 1914; Swarth 1914; Grinnell and Miller 1944; Hunter 1984). There were various estimates of the number pairs along the LCR in the 1980s; Hunter (1984) estimates there to be less than 100 individuals outside of the Bill Williams River Delta, and Serena (1981) estimates there to be 50 to 55 pairs along the main stem of the LCR and an additional 15 to 20 pairs at the Bill Williams River Delta.

Surveys for gilded flickers along the LCR have been intermittent over the years. Through historical expeditions, breeding pairs have been observed within the Lower Colorado River Multi-Species Conservation Program (LCR MSCP) planning area (Brown 1904; Cooper 1870 *in* Grinnell 1914). Surveys to document bird use along the LCR occurred in the 1970s and 1980s when breeding gilded flickers were documented along the LCR and where the above population estimates were obtained (Serena 1981; Hunter 1984; Rosenberg et al. 1991).

After the 1980s, bird surveys along the LCR have been limited and sporadic. Some areas were surveyed during other standardized surveys such as the Christmas Bird Count, the Arizona Breeding Bird Atlas, BBS, and the USFWS refuge surveys (Neale et al. 1994; Lynn and Averill 1996; National Audubon Society 2002; Corman 2005; K. Blair 2012, unpublished data; USGS Patuxent Wildlife Research Center 2012). These were general bird surveys, and even if a gilded flicker was detected, the observer often did not have time to find a nest or gather evidence of confirmed breeding. Gilded flickers probably have large home ranges similar to northern flickers, so if an individual is detected, it could be well outside their nesting territory (Royall, Jr. and Bray 1980; Elchuck and Wiebe 2003b). As mentioned above, there were only a few breeding confirmations near the Mohave and Mineral Washes on the Bill Williams National Wildlife Refuge (Neale et al. 1994; eBird 2013). The Bureau of Reclamation (Reclamation) has funded system-wide surveys for the southwestern willow flycatcher since 1998 and the yellow-billed cuckoo since 2005. Gilded flickers have been detected during the surveys, but surveyors often did not have time to confirm breeding or find the nest (McKernan and Braden 2002; Halterman et al. 2009; McLeod and Koronkiewicz 2009, 2010; McNeil et al. 2010; McLeod and Pellegrini 2011; McNeil et al. 2012).

Reclamation has funded system-wide general bird surveys that have taken place annually within the LCR MSCP planning area since 2007 (GBBO 2011, 2012). There have been several survey plots at various places at the Bill Williams River Delta and along the LCR where they were detected in the 1970s and 1980s (GBBO 2011, 2012). It appears that the population present along the LCR and at the Bill Williams River Delta in the 1970s and 1980s is largely gone except for a few isolated pairs (GBBO 2011, 2012).

To adequately determine distribution and quantity of remaining pairs (if any) breeding along the LCR, it seems system-wide surveys, specifically for gilded flickers, should be implemented (B. Sabin 2012, personal communication). As of now, the estimated population of gilded flickers breeding along the LCR and at the Bill Williams National Wildlife Refuge remains close to zero (GBBO 2011, 2012).

Gilded flickers are more often detected in the riparian habitat along the LCR as family groups or during their non-breeding season (National Audubon Society 2002; GBBO 2011, 2012; eBird 2013). There are no quantitative data on local movements of gilded flickers, but from general observations, it appears that gilded flickers are breeding in the saguaro habitat adjacent to the LCR and the Bill Williams National Wildlife Refuge, and then some are moving into the riparian habitat during their non-breeding season.

## Hybrid Populations

Some gilded flickers historically present along the LCR have had some red coloration in the wings, shafts, and tail. Short (1965) felt that those individuals represented an isolated hybrid population between *Cafer and Chrysoides*. Where hybridization originally occurred along the river is unclear, although it may have been near the Utah/Nevada border. Phillips et al. 1964 felt those individuals with red coloration did not represent introgression between *Cafer and Chrysoides* but a red variant of *Chrysoides* due to some physiological condition. The San Diego Natural History Museum has specimens from the area of Bard and Laguna Dam, of which nine of the individuals have more red than yellow in the wings, shafts, and tail (P. Unitt 2012, personal communication). The question of whether these individuals are hybrids or a red variant remains unresolved.

Recent hybrids or red variants have been sighted in the population near Cima Dome (B. Sabin 2012, personal communication). In Arizona, hybrid individuals are found in riparian zones (Short 1965). More research may need to be done to figure out if these are hybrids or red variants as along the LCR.

### Populations Adjacent to the LCR MSCP Planning Area

A small breeding population of gilded flickers has existed in the Mohave National Preserve, in San Bernardino County, California, near Cima Dome (Grinnell 1914; Grinnell and Miller 1944; Hunter 1984). East of the main stem of the LCR and south of the Bill Williams River in southern Arizona, a population of gilded flickers exist and are most abundant in saguaro habitat (Corman 2005).

## LIFE HISTORY

### General Description

Gilded flickers are large woodpeckers, measuring 28–31 centimeters (cm) in length and weighing 111 grams. 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. Males have a red malar stripe, which is lacking in females. Gilded flickers can only be confused with other flickers. They have the pale brown head of a red-shafted flickers but the yellow wings and tail of yellow-shafted flickers.

### Vocalizations and Sounds

Flickers produce an array of sounds, all for specific situations. Both sexes produce all vocalizations. Gilded and northern flicker vocalizations are essentially identical, with gilded flickers averaging a higher pitch (Sibley 2000). Flicker vocalization research is mostly associated with northern flickers. Because gilded and northern flicker vocalizations are essentially the same, most of the following research on vocalizations and sounds are based on northern flickers (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) suggests the incessant calling is a method used by the adults to recognize their fledgling young.

The most common call produced by the adults 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 spring, during pair formation and territory establishment, and continue this vocalization into fall migration (Duncan 1990; Short 1982). Observations have shown that northern flickers begin to produce the long call in February (Brackbill 1957; Royall, Jr. and Bray 1980).

Adult flickers also produce the single note “peah” vocalizations given by fledglings. The function of the “peah” is unclear. Short (1965, 1982) and Kilham (1983) consider the “peah” an alarm call, but Lawrence (1966), Burns (1900), and Duncan (1990) 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. Royall, Jr. and Bray (1980) observed northern flickers beginning to make the “peah” call early in March.

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 1965, 1982). This call is complex, variable, and poorly understood. Adult flickers also make non-vocal drumming and tapping sounds with their bills. They produce the drumming with rapid, even blows of the bill on a resonating object. Drumming usually occurs in conjunction with the long call as a territorial defense (Lawrence 1966). Tapping sounds are associated with nest excavation and food gathering (Lawrence 1966).

## Migration

Current literature suggests that gilded flickers do not migrate (Hunter 1984; Rosenberg et al. 1991).

Short (1965) found no differences in plumage and morphometrics between summer and winter gilded flickers in Baja California despite substantial geographic variation among three breeding populations, suggesting no seasonal movement between geographic populations.

## Habitat

Gilded flickers are associated with mature saguaro cacti (*Carnegiea gigantae*) in most of their breeding range (Grinnell 1914; Swarth 1914; Gilman 1915; Bent 1939; Grinnell and Miller 1944; Hunter 1984; Corman 2005). Gilded flickers that are detected in riparian woodlands within their range usually nest in saguaro cacti and forage in the riparian woodlands (Corman 2005).

Average nest cavity heights range from 1.8 to 8.86 meters in saguaro cacti (Gilman 1915; Kerpez and Smith 1990a; Corman 2005). Gilded flicker nest cavities are strongly associated with the largest saguaro cacti (Kerpez and Smith 1990a). Gilded flicker density was not correlated with density of large saguaro cacti probably because gilded flickers only rely on the cacti as nesting habitat and not as foraging habitat (Kerpez and Smith 1990a); they mostly forage on the ground or in annual foliage (Kerpez and Smith 1990a).

Along the LCR, gilded flickers are associated with cottonwood and willow riparian areas (Gilman 1909; Grinnell 1914; Serena 1981; Hunter 1984; McKernan and Braden 2002; Halterman et al. 2009; McLeod and Koronkiewicz 2009, 2010; McNeil et al. 2010, 2012; McLeod and Pellegrini 2011). If saguaros are adjacent to the riparian habitat, they are usually used as nest cavities, and the riparian area is used as foraging habitat (Grinnell 1914; Swarth 1914; Grinnell and Miller 1944; Rosenberg et al. 1982, 1991; GBBO 2011, 2012). Brush et al. (1983) found that gilded flickers prefer soft wood as nest sites, and the occurrence of gilded flickers seems to be dependent on nest site availability. Additional studies need to be conducted on the habitat preferences of gilded flickers in riparian habitat.

Typically, gilded flickers stay away from densely populated urban and rural neighborhoods (Rosenberg et al. 1991; Corman 2005). Gilded flickers are not restrained by cover at the local level but are at the landscape level. They routinely forage on open ground and turf grass (Turner 2006).

## 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 the gilded flicker’s diet is similar to that of northern flickers. Beal (1911) reports the northern flicker’s diet consisted mostly of ants (*Formica, Lasius, Campontus, Myrmica*, *Cremastogaster*, *Aphaenogaster*, *Prenolepis*, *Pheidole*, *Solenopsis*, *and Tetramorium*) and ground beetles. Beal (1911) found that in fall and winter, flicker diets shifted to fruits. Primarily, flickers forage for food on the ground, in soil, leaf litter, and in anthills, probing and hammering their bill. Flickers are rarely found foraging in trees.

Elchuk and Wiebe (2003a) found that northern flickers never forage on large ant mounds built by the aggressive *Formica* species but instead foraged on individual ants on the ground or in patches of bare ground containing numerous small ant craters. The aggressive behavior of the ant mounds appear to make to make them unpalatable prey (Elchuk and Wiebe 2003a).

# 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 their bodies. 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, Jr. and Bray (1980), using radio transmitters to monitor roosting habitats, found northern flickers arriving at the roost just before sunset and leaving it up to 25 minutes before sunrise. Royall, Jr. and Bray (1980) also observed some northern flickers using the same roost every night and other northern flickers using 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 territories; very few 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. Fisher and Wiebe (2006b) found that northern flickers defend their nests against predators. Age, sex, brood size, and body condition do not affect the intensity of nest defense (Fisher and Wiebe 2006b). Elchuk and Wiebe (2003a) found that northern flickers do not defend their feeding territory and frequently forage with neighboring northern flickers without showing any aggressive behavior. The home ranges of northern flickers frequently overlap (Elchuk and Wiebe 2003a). Kilham (1973) found that when yellow-shafted flickers begin to incubate, they stop long calling and drumming, and it is possible for a second pair to establish a nearby territory.

Elchuck and Wiebe (2003b) found that home ranges of northern flickers varied from 5 to 109 hectares (ha), with an average of 25 ha. The average size of a core area was 7 ha, and home ranges had multiple core areas, which included nest sites, foraging sites, and perching locations (Elchuck and Wiebe 2003b). The farthest distance a northern flicker flew to forage was 1,300 meters (Elchuck and Wiebe 2003b). Elchuck and Wiebe (2003b) observed that the size and shape of home ranges related to the spatial distribution of foraging hot spots. They also observed that home ranges were larger on subsequent laying attempts, and as distance to neighbor increased, so did home range (Elchuk and Wiebe 2003b). Royall, Jr. and Bray (1980) found that the winter home range of northern flickers ranged from 48 to 101 ha.

## Threats

Records exist of Harris hawks (*Parabuteo unicinctus*), sharp-shinned hawks (*Accipiter striatus*), Cooper's hawks (*A. cooperii*), and broad-winged hawks (*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).

European starlings (*Sturnus vulgaris*) are common nest cavity competitors with flickers (Bent 1939; Ingold 1994, 1996); however, they may not compete with gilded flickers (Kerpez and Smith 1990a). Kerpez and Smith (1990a) studied competition between gilded flickers and European starlings for nest cavities in saguaros around Tucson, Arizona. They found that European starlings do not compete with gilded flickers. They attribute this to the larger size of gilded flickers and their 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 northern flicker nests. Wiebe (2003) found that European starlings seized 7% of northern flicker nests. Ingold observed a European starling on the back of a northern flicker, pecking the flicker repeatedly.

Ingold (1994) found that at least some of the northern flickers that were affected by starlings were able to re-nest later in the season. In some locations where nest usurpation by starlings is high, northern flickers may delay nesting to avoid competition (Ingold 1996; Wiebe 2003). However, the northern flicker's clutch size decreases as their breeding season progresses (Ingold 1996; Wiebe 2003).

Northern flickers may face a nest site tradeoff between safety from predators and competitors (Fisher and Wiebe 2006a).

## 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 planning area, gilded flickers use 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 males taking a dominant 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 (1990b) analyzed gilded flicker saguaro nests in Tucson, Arizona. Nest cavity heights averaged 6.2 meters, entrance horizontal diameters averaged 8.3 cm, entrance vertical diameters averaged 7.0 cm, vertical depths averaged 37.6 cm, and horizontal depths averaged 12.5 cm. Zwartjes and Nordell (1998) found nest cavity orientation of gilded flickers to be north-northeast; however, Kerpez and Smith (1990b) found nest cavity orientation to be random. Zwartjes 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 using the holes studied.

The dimensions of gilded flicker cavities are different than Gila woodpecker cavities (Kerpez and Smith 1990b). Kerpez and Smith (1990b) found no difference in the height of cavities between the two species, but McAuliffe and Hendricks (1988) found that gilded flicker cavities were placed in saguaro cacti at significantly higher heights than Gila woodpecker cavities.

Corman (2005) reports gilded flickers occupying nests as early as March 12, with young observed on April 3. The latest observed young in a nest was on July 2 (Corman 2005). The gilded flicker’s 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 attempts fail. 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 takes usually 11 days. The nestlings fledge between 21–27 days old. Adult flickers may decrease feeding the young and use “peah” and long calls to entice hatchlings to leave the nest. Juvenile dependence on adults is unknown but presumed to be short (Moore 1995). Wiebe (2005) found that northern flickers have about a 13% chance of pairing with the same mate from the previous year.

Wiebe (2002) reported a polyandrous female northern flicker attending two nests; one nest was in the incubation stage, while the other nest was in the nestling stage. All nestling in both broods successfully fledged.

### Demography

First breeding for flicker males occurs the spring following birth. First breeding for females is unknown but likely to be the spring following birth as well (Moore 1995).

Based on banding records, flickers can live at least 9 years and 2 months (Clapp et al. 1983). Survivorship records do not exist.

## NEST BOX/SNAG INSTALLATION

There are no data on gilded flicker using installed nest boxes or snags. Data on nest dimensions are limited. Kerpez and Smith (1990a) analyzed gilded flickers saguaro nests in Tucson, Arizona. Nest cavity heights averaged 6.2 meters, entrance horizontal diameters averaged 8.3 cm, entrance vertical diameters averaged 7.0 cm, vertical depths averaged 37.6 cm, and horizontal depths averaged 12.5 cm.

Bower (1995) successfully attracted northern flickers in multiple years to a nest box he created outside his home in Michigan. The nest box was 76 cm tall with a 16.0 x 18.0 cm floor and a 6.4-cm entrance hole, and filled to the brim with planar shavings. He made the floor of the box rounded to simulate a natural cavity and put kerfs in the inside sides of the box to give the fledglings something to cling to (Bower 1995).

Ingold (1998) placed 54 nest boxes near active northern flicker cavities to try to reduce nest site competition with European starlings. Only one northern flicker pair opted to use the nest cavity instead of the natural cavity (Ingold 1998). Ingold (1998) felt that the presence of nest boxes may have attracted additional starlings to the northern flicker breeding area.

## 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.

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# Least Bittern (*Ixobrychus exilis*)

## INTRODUCTION

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

## LEGAL STATUS

Least bitterns are listed as a species of special concern in Arizona and California (Remsen, Jr. 1978; Arizona Game and Fish Department 2001). They are listed by the U.S. Fish and Wildlife Service (USFWS) as a migratory non-game bird of conservation concern (USFWS 2008). They are on the Audubon Society's Blue List, which is published in their ornithological field journal *American Birds* (Tate 1986). Least bitterns are a protected species in Nevada (Department of Conservation and Natural Resources, Nevada Natural Heritage Program 2010). They are not listed by Mexico.

## DISTRIBUTION

### Historical Range

Bent (1926) lists the breeding range of least bitterns 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 least bitterns in Arizona was one occurrence along 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 (LCR) (Monson 1949). Monson and Phillips (1981) list least bitterns as fairly common residents in the Colorado River Valley, north to Topock, and as residents in the cattails (*Typhus* spp.) along the Salt River in the Phoenix area. In California, Grinnell (1915) list them as fairly common summer visitants north through the interior of the Sacramento Valley and also occurring along the southern coast. In 1940 and 1941, this species was 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) lists least bitterns as infrequent in summer and reports them from Washoe, Elko, Churchill, Esmeralda, and Clark Counties. The first records for southern Nevada were from Lake Mead, September 1938 (Grater 1939). Least bitterns were recorded from the Great Salt Lake in the 1880s (Ryser, Jr. 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 least bitterns is now from southeastern Canada, through the United States and Mexico, to Costa Rica (American Ornithologists' Union 1998). They are 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 the 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. 2013). Breeding has recently been confirmed in central, south-central, and southeastern Arizona as well as southern Nevada (Corman 2005; Branca 2005).

Their 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 the LCR MSCP Planning Area

Rosenberg et al. (1991) state that the largest populations along the LCR are in extensive cattails and bulrush (*Scirpus* spp.) marshes, such as at Topock Marsh and near Imperial Dam. The Arizona Breeding Bird Atlas states that, in much of southwestern Arizona at elevations below 1000 feet (305 meters), extensive cattail marshes often harbor least bitterns (Corman 2005). In 2006, presence/absence surveys for Yuma clapper rails (*Rallus longirostris yumanensis*) along the LCR were modified to include surveys for least bittern, California black rails (*Laterallus jamaicensis coturniculus*), and Virginia rails (*R. limicola*) (USFWS 2006).

In Reaches 1 and 2, least bitterns are listed as rare transients in the Lake Mead National Recreation Area (National Park Service 1978). One was heard in June 2005 near the inflow of the Muddy River into Lake Mead (J. Barnes 2006, personal communication).

In Reach 3, the majority of marsh habitat is located in Topock Marsh, Topock Gorge, and the Bill Williams River Delta. Least bitterns have been detected during marsh bird surveys at the Big Bend Conservation Area and Laughlin Bay (table 1). The bird checklists for the Havasu and Bill Williams River National Wildlife Refuges all list least bitterns as common, certain to be seen in suitable habitat from June through August, and nest locally (USFWS 2012a, 2012b).

Table 1.—Least bittern survey data (USFWS 2014)

| Survey year | 2006 | 2007 | 2008 | 2009 | 2010 | 2011 | 2012 | 2013 |
|---|---|---|---|---|---|---|---|---|
| Reach 1 | NS[1] | NS | NS | NS | NS | NS | NS | NS |
| Reach 2 | NS | NS | NS | NS | NS | NS | NS | NS |
| Reach 3 | 117 | 36 | 84 | 82 | 107 | 91 | 80 | 65 |
| Reach 4 | 8 | 112 | 61 | 55 | 24 | 60 | 88 | 51 |
| Reach 5 | 197 | 138 | 111 | 140 | 108 | 144 | 68 | 79 |
| Reach 6 | 107 | 85 | 61 | 93 | 52 | 47 | 63 | 52 |
| Reach 7 | 1 | NS | NS | NS | NS | NS | 1 | NS |

[1] NS = No survey.

In Reaches 4 and 5, there is scattered marsh habitat through the Colorado River Indian Tribe (CRIT) Reservation, Cibola and Imperial National Wildlife Refuges, and near Imperial Dam (see table 1). The USFWS lists least bitterns as nesting in the Cibola and Imperial National Wildlife Refuges (USFWS 1974, 1994). In Reach 6, the only extensive marsh habitat is at Mittry Lake (table 1). Reach 7 contains very little habitat for least bitterns (see table 1).

# LIFE HISTORY

## General Description

Least bitterns are the smallest members of the heron family and are one of the most inconspicuous of all marsh birds (Weller 1961; Gibbs et al. 1992). Their very small size (28–36 centimeter [cm] length, 43-cm wing span, 80-gram 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. The legs and feet are a straw to buffy yellow. The 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, Jr. 1975).

Males vocalize a dove-like “coo” that is repeated five 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) describe them as locally common breeders 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* and 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). The distance from open water can range from 60 cm to 65 meters but is usually less than 10 meters (McVaugh, Jr. 1975; Aniskowicz 1981; Gibbs et al. 1992). In Canada, least bitterns were detected breeding only in wetlands ≥ 6.8 hectares (ha) in size and ≥ 25.2 cm in depth (Tozer et al. 2010).

Nests have been found scattered throughout suitable habitat or concentrated in loose groupings where the distance between them ranged from 2 to 85 meters (McVaugh, Jr. 1975; Ziebell 1990). Nests have been recorded 1 meter apart in highly productive habitat where least bitterns 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 (*Quiscalus mexicanus*) colonies in southern Nevada (Post and Seals 1993; J. Healy 2006, personal communication).

Typical clutches are four to five eggs (range of two to seven), 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).
Least bitterns will re-nest and double brood (Post and Seals 1993; Bogner and Baldassarre 2002).

## Diet

The least bittern's major food items are small fish and insects (Gibbs et al. 1992). The stomach contents of 20 least bitterns was collected at the Imperial National Wildlife Refuge 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. 1992; J. Healy 2006, personal communication).

The least bittern's small size, highly compressed trunk, and ability to grasp with its feet enable it to move through dense vegetation. They forage by clinging to emergent vegetation over open water and extending their long necks, 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 least bitterns are standing in one place, walking slowly, moving their heads back and forth, and flicking their wings to startle prey (Kushlan 1978).

# Habitat

In general, least bitterns occupy 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 cattails (*Typha domingensis*) and broad-leaved cattails (*T. latifolia*), but some may use common reed (*Phragmites australis*) or salt cedar (*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 2). Some sawgrass (*Cladium jamaicense*) and cattail habitat were 2 meters in height (Kushlan 1973; McVaugh, Jr. 1975).

Table 2.—Depth of water and vegetation type at least bittern nest sites

| Depth of water (cm) | Vegetation type | Author(s) |
|---|---|---|
| 40 | Cattail | Aniskowicz 1981 |
| 60 | Cattail | Nero 1950 |
| 12–40 | Cattail | Post and Seals 1993 |
| 8–97 | Cattail and bulrush | Weller 1961 |
| 30 | Cattail | McVaugh 1975 |
| 5–29 | Cattail | Manci and Rusch 1988 |
| 25–65 | Cattail | Tozer et al. 2010 |
| 50 | Dense vegetation | Fredrickson and Reid 1986 *in* Gibbs et al. 1992 |

Least bitterns are 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 per ha in a 13-ha study area and 12 pairs per hectare 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 re-nests or second nests. The breeding density of least bitterns in some marshes along the LCR has been estimated at 40 birds per 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 (1992) state that the preservation of wetlands greater than 5 ha with dense, tall (> 1 meter) emergent vegetation over relatively deep water (10–50 cm) and interspersed with patches of open water is the most important management need for least bitterns. In Canada, the abundance of least bitterns increased dramatically in wetlands greater than 45 cm deep (Tozer et al. 2010).

## Threats

Least bitterns 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 their 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 LCR may be bioaccumulating potentially toxic concentrations of selenium in their tissues and eggs (King et al. 2000). Least bitterns collected at the Imperial National Wildlife Refuge had selenium concentrations in their livers above the 3-parts-per-million threshold for which toxic effects on predatory fishes or birds might be expected (Martinez 1994).

Destruction of wetland habitat is likely the greatest threat to least bitterns 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 of habitat provided by marshes associated with backwaters. Up to 28.3 ha of habitat could be removed to maintain channel functions (e.g., dredging desilting basins) in the river and 12.1 ha in irrigation ditches (Bureau of Reclamation 2004).

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# MacNeill's Sootywing (*Hesperopsis gracielae* MacNeill)

## LEGAL STATUS

MacNeill's sootywings (*Hesperopsis gracielae* MacNeill) are skipper butterflies. The species was previously a Federal category 2 candidate and is currently listed as a species of concern. In Nevada, they are listed as S1 (critically imperiled) (Department of Conservation and Natural Resources, Nevada Natural Heritage Program 2016). In California, the skippers are listed as S1 (extremely endangered) and S3 (restricted range, rare) (California Department of Fish and Game 2002). They do not have State protection in Arizona (Arizona Game and Fish Department 2003). They are managed as a sensitive species by both the U.S. Forest Service and the Bureau of Land Management.

## DISTRIBUTION

### Historical Range

The MacNeill's sootywing's historical range was along the lower Colorado River (LCR) and near the river along its tributaries in southeastern California, western Arizona, southern Nevada, and southern Utah (Scott 1986). Their type locality is along the California side of the LCR, near Parker, Arizona.

### Current Range

The current range of MacNeill's sootywings approximates their historical range, except their presence in southern Utah is uncertain. MacNeill's sootywing populations have been detected during sweep-net collections of butterflies along the LCR (Nelson and Andersen 1999).

### Populations Within the LCR MSCP Planning Area

MacNeill's sootywings have been observed in the following areas:

- Big Bend Conservation Area and parcels south of Davis Dam, Laughlin, Nevada, and Bullhead City, Arizona

- Havasu National Wildlife Refuge (Lost Lake revegetation site and Beal Lake Conservation Area), Needles, California, and northwest of Needles

- Bill Williams River National Wildlife Refuge near Lake Havasu, Arizona

- Colorado River Indian Reservation (No-Name Lake revegetation site)

- Palo Verde Ecological Reserve (Phases 4 and 6), Blythe, California, and on parcels northeast of Blythe, California.

- Cibola National Wildlife Refuge (Cibola NWR) (including Unit B revegetation sites and the Island Unit revegetation site), south of Blythe, California, and also observed near the Cibola NWR (1978 Dredge Spoil revegetation site)

- Cibola Valley Conservation Area (Phases 4 and 5)

- Hart Mine Marsh, La Paz County, Arizona

- Hunters Hole, Gadsden, Arizona

# LIFE HISTORY

## General Description

MacNeill's sootywings are small (wingspread = 23 millimeters) skippers with dark brown and black mottled wings (MacNeill 1970). Skippers are butterflies in the family Hesperiidae, named after their quick, darting flight habits. Skippers have stout bodies, widely separated wing venation, and antennae clubs that are hooked backwards.

Sootywing eggs are described as spherical and heavily sculptured with a ridged chorion and are reddish brown in color (Wiesenborn and Pratt, 2008). However, the eggs are described as ivory-white in color by Emmel and Emmel (1973). Pratt and Wiesenborn (2011) further indicate that sootywing eggs from the Cibola NWR north to Overton, Nevada, are dark in color, while those from the Imperial National Wildlife Refuge south to Baja California are a cream or ivory color.

Caterpillars are described as blue to yellow green, covered with tiny white dots, with the head black and heart shaped. The head is covered with short, white hair (Allen et al., 2005) and is large relative to the thorax, creating a constriction between the head and the first segment. Caterpillar nests (unlike those of some moth caterpillars) are devoid of droppings (Allen et al., 2005). Sootywing caterpillars are foliage feeders, and leaf damage often occurs only on the surface portion of quailbush (*Atriplex lentiformis*) leaves.

## Life Cycle

MacNeill's sootywings deposit ivory-white or reddish-brown colored spherical eggs singly on quailbush leaves. The larvae cut and wrap a leaf around themselves, securing the leaf with silk produced near their mouths. The insect pupates within the wrapped leaf and likely overwinters as pupae. Two or three adult flights occur per year (MacNeill 1970).

## Feeding

Larvae only survive to adulthood feeding on quailbush. It is unknown if nectaring is necessary for adult survival. Nectaring is not a commonly observed sootywing behavior. Pratt and Wiesenborn (2009) observed a large number of sootywing behaviors (1,620) and noted that 12% of these behaviors involved landing on flowers (nectaring).

## Habitat

MacNeill's sootywings require dense stands of quailbush, ideally mixed with nectar-producing plants (specificity unknown), but sootywings have been found in areas without nectar-producing plants. Sootywings require quailbush that contains high leaf water content resulting from shallow groundwater or precipitation events.

## Threats

The species was originally listed as a 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, lowered water tables, and conversion of habitat to agriculture. Along with xerification, the greatest present threat likely is destruction of habitat for housing development. An additional threat may be replacement of native quailbush with cultivated varieties.

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# Relict Leopard Frog (Rana = *Lithobates onca*)

## LEGAL STATUS

Platz (1984) suggests that relict leopard frogs (*Rana onca*) should be considered extinct. The last known specimen of the species was seen in Utah in the 1950s and is believed to be extirpated in the State. They were rediscovered in 1991 in parts of their historical range (Black Canyon/Virgin River) through southern Nevada and northwestern Arizona (Bradford et al. 2004). Seven populations of relict leopard frogs were found in three distinct areas (Bradford et al. 2004). They were listed as a candidate species on May 4, 2004 (U.S. Fish and Wildlife Service [USFWS] 2004). A petition to list them as an endangered species was received by the USFWS on May 9, 2002 (USFWS 2004). NatureServe, a nonprofit organization that ranks species on their relative imperilment, ranked relict leopard frogs as critically imperiled across their 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). Relict leopard frogs are listed as a species of special concern in Arizona.

## DISTRIBUTION

### Historical Range

Relict leopard frogs 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 (Relict Leopard Frog Conservation Team [RLFCT] 2005; Jaeger et al. 2001; Bradford et al. 2004). They were 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. The species was historically found in Utah from the vicinity of Hurricane, Washington County, downstream through the Virgin River Valley, at elevations between 370 and 760 meters (Center for Biological Diversity and Southern Wilderness Alliance 2002). Historical abundance is unknown (Bradford and Jennings 2005).

The Arizona Game and Fish Department (AGFD) conducted surveys within the historical range of relict leopard frogs in Arizona. Limited surveys were conducted before 1997 in which the species was detected at one site near Littlefield, Arizona. The surveys were conducted from February to November in the years 2001, 2002, and 2003 at 74 locations; no relict leopard frogs were found. Six of the 74 sites fit the 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 relict leopard frogs were found. The species was rediscovered at Corral and Bluepoint Springs in 1991. Bradford et al. (2004) conducted targeted surveys for relict leopard frogs at 66 locations across their historical range between 1991 and 2001; conducted visual estimate and mark-recapture surveys on extant populations of the species between 1991 and 2001 to determine population size and structure; 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 conducted visual encounter surveys at Reber Spring from 1998 to 2001. They also conducted mark-recapture studies at Blue Point Spring in 1995 and 1996 and at Bighorn Sheep Spring in 2001.

## Current Range

Extant populations of relict leopard frogs 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 kilometers 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 kilometers in length), and (3) adjacent to the Virgin River 4 kilometers northeast of Littlefield, Mohave County, Arizona (Reber Spring) (Bradford et al. 2004; Center for Biological Diversity and Southern Wilderness Alliance 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 (*R. catesbeiana*) (Bradford et al. 2004). Bradford et al. (2004) estimated that the total number of frogs at all sites, based on mark-recapture data, visual encounter surveys, and extent of habitat, was at least 1,077 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. Through the mark-recapture study at Blue Point Spring in 1996–97, it was estimated that relict leopard frogs had an annual survival rate of 27% for those years (Bradford et al. 2004). The USFWS estimated that the current distribution is less than 20% of their historical distribution (USFWS 2004). As of 2012, there were a total of 6 natural sites where relict leopard frogs still occur and 11 experimental sites where the frogs have been translocated (figure 1), with more than one-half of the estimated population occurring within the experimental translocation sites (Jaeger and Rivera 2013).

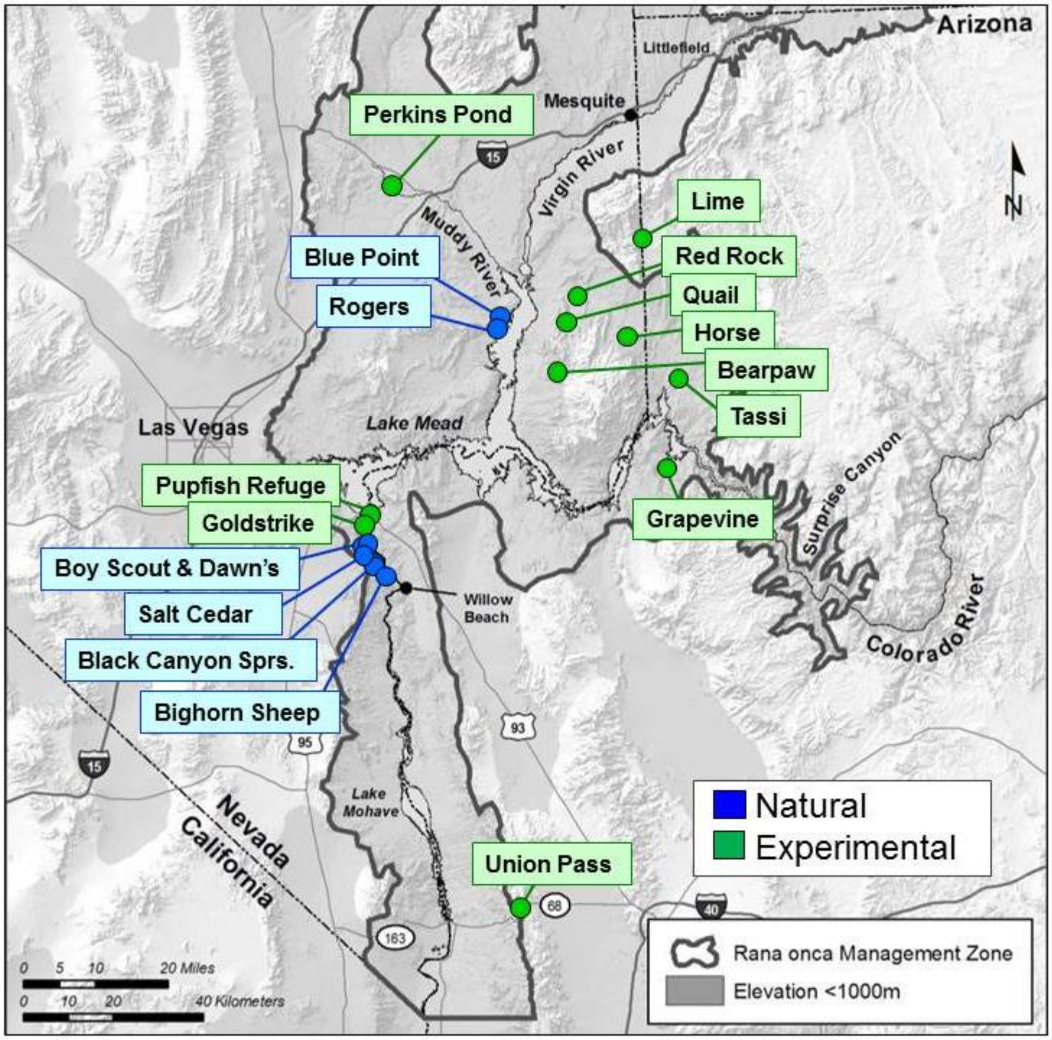

**Figure 1.—Locations of sites containing natural populations of relict leopard frogs (in blue) and sites containing active experimental translocation sites (in green) (Jaeger and Rivera 2013).**

# LIFE HISTORY

## General Description

Henry Crecy Yarrow collected the first specimen of relict leopard frogs 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. Relict leopard frog are true frogs (family Ranidae) in the *R. pipiens* complex (leopard frogs) (Jennings 1988). The following traits distinguish them from other species in the *R. 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) the 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; Amphibia Web 2005; NatureServe 2005). Males are less spotted, more uniform in color, and are smaller in size than females (Jennings 1988). Fully developed tadpoles reach 85 millimeters in length and have a greenish olive dorsum, a heavily mottled pale green-yellow tail, and a light venter (Amphibia Web 2005). Wright and Wright (1949) describe the call of relict leopard frogs to be shorter than and not as loud as that of *R. pipiens* (Platz 1984).

## Breeding

Relict leopard frogs breed 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 (AGFD 2003; Center for Biological Diversity and Southern Wilderness Alliance 2002; NatureServe 2005; Bradford and Jennings 2005). Oviposition is known to vary among sites (Bradford and Jennings 2005). Relict leopard frogs deposit egg masses in clusters of up to 250 eggs (AGFD 2003; Center for Biological Diversity and Southern Wilderness Alliance 2002; NatureServe 2005; Bradford and Jennings 2005). Males reach reproductive maturity at 42 millimeters in length measured from snout to vent (Bradford and Jennings 2005). Malfatti (1998) was able to create conditions that allowed relict leopard frogs to breed in captivity. The life history of the species, especially in regard 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 and Southern Wilderness Alliance 2002; NatureServe 2005).

## Habitat

Relict leopard frogs inhabit permanent streams, springs, and spring-fed wetlands below 720 meters in elevation that have constant water temperatures between 16 and 55 degrees Celsius (Bradford et al. 2004; Center for Biological Diversity and Southern Wilderness Alliance 2002). Historic relict leopard frog habitat includes sites with some submerged, emergent, or perimeter vegetation that supports an adequate amount of food resources (Center for Biological Diversity and Southern Wilderness Alliance 2002; Jennings and Hayes 1994). Lentic systems at least 400 square miles in area and lotic systems at least 400 meters in length with a depth of 1 meter are defined as suitable habitat by the AGFD.

Habitats must have adequate heterogeneity to provide cover and oviposition sites and be free of non-native predators (Blomquist et al. 2003). Platz (1984) describes the relict leopard frog's historical habitat as sites with permanent cold water and stream pools that are 12 to 16 inches deep.

The five sites currently inhabited by relict leopard frogs are characterized by spring systems with largely unaltered hydrology and no introduced American bullfrog (*R. catesbeiana*) or game fishes (Bradford et al. 2004). Excessive emergent vegetation of native and non-native species is believed to be a threat to relict leopard frogs (RLFCT 2005). Bradford and Jennings (2005) observed that adults prefer relatively open shorelines where dense vegetation does not dominate. Vegetation encroachment was believed to be the major cause of the species being extirpated from Corral and Reber Springs (Bradford and Jennings 2005). A habitat study at Blue Point Spring using radio telemetry found that frogs favored areas of the spring that were less densely vegetated (Harris 2006). Habitat manipulation experiments have been conducted in order to create more open areas that may increase frog use. Observational data suggest that these areas do encourage frog use, and egg masses have been found in these areas (J. Jaeger 2005, personal communication).

## Movement

Relict leopard frogs are primarily nocturnal during the summer months and switch to a diurnal schedule during winter. There is no evidence of torpor, hibernation, or migration of the species (Center for Biological Diversity and Southern Wilderness Alliance 2002). Relict leopard frogs are inactive in cold temperatures (AGFD 2003). They are restricted to narrow habitat corridors and will not move across desert habitats (Center for Biological Diversity and Southern Wilderness Alliance 2002).

## The Relict Leopard Frog as Distinct Taxon

Since their rediscovery, there have been questions about whether populations of leopard frogs in the Black Canyon/Virgin River area are relict leopard frogs, lowland leopard frogs (*R. yavapaiensis*), or a hybrid of relict leopard frogs and lowland leopard frogs. Jaeger et al. (2001) used mitochondrial deoxyribonucleic acid (mtDNA) and morphological analyses to determine that the seven populations of leopard frogs in the Black Canyon/Virgin River area were one distinct taxon (the relict leopard frog) (Jaeger et al. 2001). They recommended that conservation strategies that retain leopard frogs in the Black Canyon/Virgin River (the relict leopard frog) as evolutionary distinct units be developed. The results of an additional genetic study confirmed that relict leopard frogs and lowland leopard frogs are distinct taxa (Oláh-Hemmings et al. 2010). The systematic relationship between extinct Vegas Valley leopard frogs

(*R. fisheri*) and relict leopard frogs remains unresolved. Some authors believe that Vegas Valley leopard frogs (*R. fisheri*) and relict leopard frogs are synonyms. Jennings et al. (1995) concludes that Vegas Valley leopard frogs are not synonymous with relict leopard frogs and should be considered a valid, distinct species (Center for Biological Diversity and Southern Wilderness Alliance 2002). More recently, an archival deoxyribonucleic acid (DNA) analysis found that Vegas Valley leopard frogs are more closely related to threatened Chiricahua leopard frogs (*R. chiricahuensis*) than to relict leopard frogs (Hekkala et al. 2011).

## Reasons for Decline and Threats

Relict leopard frogs have declined across their range and are vulnerable to extinction. The remaining habitat in which the species occurs and their 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 relict leopard frogs 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 fishes, crayfish, western spiny soft-shell turtles) (*Apalone spinifera*) (Bradford et al. 2004; Center for Biological Diversity and Southern Wilderness Alliance 2002; Jennings 1988; Moyle 1973; Platz 1984). Water development has historically flooded relict leopard frog habitat, eliminated brief annual floods that would prevent the encroachment of emergent vegetation, and eliminated connectivity between remaining populations (Center for Biological Diversity and Southern Wilderness Alliance 2002). Agricultural and urban development have destroyed their habitat (Center for Biological Diversity and Southern Wilderness Alliance 2002). Extensive grazing can cause habitat degradation, although in some situations, managed grazing of emergent vegetation may benefit relict leopard frogs by providing open water habitats (Center for Biological Diversity and Southern Wilderness Alliance 2002).

Threats to extant populations of relict leopard frogs 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 and Southern Wilderness Alliance 2002; NatureServe 2005; RLFCT 2005). Hayes and Jennings (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 fishes and habitat alteration, are equally responsible. Non-native fishes are recognized as a threat to relict leopard frog populations. Observations have been made that provide the basis for the hypothesis that non-native fishes reduce eggs and tadpole survival, thus limiting recruitment into adult populations at the Northshore area springs. A proposed study would determine whether introduced fishes can be removed from a section of Blue Point Spring by use of fish barriers and common eradication techniques. The study will involve testing the hypothesis that eradication of non-native fishes increases egg and tadpole presence and metamorph-juvenile frog recruitment from manipulated segments of Blue Point Spring (Jaeger and Riddle 2005).

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# Sonoran Yellow Warbler (*Setophaga petechia sonorana*)

## LEGAL STATUS

Sonoran yellow warblers (*Setophaga petechia sonorana*) are not a federally protected species. In California, they are listed as a species of special concern (breeding), Priority 2, and have not been included on prior special concern lists (Remsen, Jr. 1978; California Department of Fish and Game 1992).

## DISTRIBUTION

### Historical Range

Swarth (1914) characterizes Sonoran yellow warblers as common summer residents confined almost entirely to the lower Sonoran River valleys, including the Colorado River Valley from Ft. Mohave to Yuma, Arizona. They were formerly common along major rivers such as the Rio Grande and lower Colorado (Lowther et al. 1999). Given the great expanse of cottonwood-willow habitats (*Populus* spp., *Salix* spp.) at the turn of the 19th century, Rosenberg et al. (1991) speculated that the total size of the Sonoran yellow warbler population in the lower Colorado River Valley was “enormous” at that time. In 1914, an estimated one to four males occurred in every 0.40 hectare of cottonwood-willow habitat along the river (Grinnell 1914). In the 1940s, Sonoran yellow warblers were “abundant” breeders along the entire California side of the lower Colorado River Valley below 183 meters (Grinnell and Miller 1944). Populations in California have declined throughout State, since the 1930s, with the loss of breeding habitat. A steeper rate of decline has occurred since the 1950s (Small 1994). Rosenberg et al. (1991) reported that Sonoran yellow warblers bred “commonly” along the Bill Williams River and at Topock Marsh in 1952 but had disappeared from these historic nesting sites after 1955. They were considered extirpated from the valley by 1960 (Monson and Phillips 1981). In the late 1970s and early 1980s, sporadically singing males were reported along the California side of the lower Colorado River (LCR), and, in 1986, one female was observed feeding a juvenile near Blythe (Garrett and Dunn 1981; Rosenberg et al. 1991; Hunter 1984). Since the 1960s, Sonoran yellow warblers have been extirpated as breeders 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 Sonoran yellow warblers includes the southwestern portion of the United States into northern Mexico. By all known accounts, populations of the species have fluctuated since their period of high abundance in the early 1900s. Despite population changes and local extirpations, the overall range of Sonoran yellow warblers today has changed little since 1944 (Heath 2008). When considering historical cottonwood-willow habitat along the LCR prior to the 1900s, Sonoran warbler numbers are likely far below historical population levels (Rosenberg et al. 1991). Reports of total extirpation along the LCR (Small 1994) possibly reflected localized extirpations rather than river-wide losses (McKernan and Braden 2002). More recently, Sonoran yellow warblers have been found in greater abundance along the LCR, the Virgin River, and the Bill Williams River (Great Basin Bird Observatory 2012, 2013, 2014). Along the LCR in Mexico, they have been detected as migrants, but very few have been found during their breeding season (Hinojosa-Huerta et al. 2008).

**Figure 1.—Distribution of yellow warblers (*aestiva* group), which includes Sonoran yellow warblers. Individuals of this group also winter in northern South America (from Lowther et al. 1999).**

## Winter Range

Yellow warblers are 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 February 7 to March 7, 1951 (Phillips et al. 1964).

## Populations Within the LCR MSCP Planning Area

Sonoran yellow warblers were common breeding birds along the LCR but were considered extirpated as a breeding species in the Lower Colorado River Multi-Species Conservation Program (LCR MSCP) planning area by the mid-1950s (Rosenberg et al. 1991). In 2007, system-wide avian surveys began along the LCR using the double-sampling method. There have been confirmed breeding pairs found along the river from Hoover Dam to the Cibola National Wildlife Refuge. Sonoran yellow warblers have also been found in large abundance along the Virgin and Bill Williams Rivers. Estimates for the number of pairs present along the rivers ranged from 700 to 1,300 from 2011 to 2013 (Great Basin Bird Observatory 2012, 2013, 2014). This species has also been found recently in areas being restored with native cottonwood and willow. Confirmed breeding pairs have been found at several LCR MSCP sites, including the Cibola Valley Conservation Area, Palo Verde Ecological Reserve, Cibola National Wildlife Refuge Nature Trail, and Beal Lake Conservation Area.

# LIFE HISTORY

## General Description

Yellow warblers are medium-sized, foliage-gleaning wood warblers (12–13 centimeters long; 9–11 grams). The plumage is more extensively yellow than most other wood warblers and are unique in having yellow on the inner webs of the tail feathers, except for the middle pair. Yellow warblers have indistinct wing bars. 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. Yellow warblers are a widespread species in North America, breeding as far north as the tundra regions of Canada (Lowther et al. 1999). Yellow warblers remain common in much of their range as habitat generalists.

A recent taxonomic review listed 43 recognizable subspecies, arranged into 3 groups mainly based on the adult male’s 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. Sonoran yellow warblers breed only along the LCR 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; Hinojosa-Huerta et al. 2008; Lowther et al. 1999, Sauer et al. 2014).

The *aestiva* group male yellow warbler in breeding plumage has his face, throat, and remaining underparts bright yellow 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 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 and more greenish, streaking on underparts is reduced or lacking, and they have a whitish or pale yellowish eye ring (Lowther et al. 1999).

## Breeding

Historically, Sonoran yellow warblers bred in the willow and cottonwood habitats that lined the LCR. Basic life history traits, such as number of nesting attempts and extent of double brooding adult or juvenile survivorship rates, are unreported. Monitoring Avian Productivity and Survivorship banding and color banding of this species takes place at several LCR MSCP sites. Through these efforts, confirmed resident breeding Sonoran yellow warblers have been found as early as April 4 and as late as the last week of September (Dodge and Kahl 2011, 2012, 2014).

## Feeding

There are no specific studies on the diet of Sonoran yellow warblers. Yellow warblers, though, are a generalist species that appear to adapt their foraging to variation in local vegetation structure (Petit et al. 1990). The yellow warbler's diet in California contained over 97% animal matter, including ants, bees, wasps, caterpillars, beetles, true bugs, flies, and spiders (Beal 1907).

## Habitat

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 *(Baccharis glutinosa)*, mesquite (*Prosopis* spp.), and salt cedar (*Tamarix* spp.) (Wise-Gervais 2005).

In surveys conducted along the LCR, Sonoran yellow warblers were found in a variety of habitat types, including mature cottonwood-willow, emergent cottonwood-willow, salt cedar, and salt cedar mixed with native vegetation (Great Basin Bird Observatory 2012, 2013, 2014).

Yellow warblers make use of salt cedar and athel tamarisk (*Tamarix aphylla*) as both a nest substrate plant and as nesting habitat along the Colorado River in the 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 salt cedar 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). On the Las Vegas Wash and at LCR MSCP sites, the species was found to colonize restored cottonwood-willow habitat within several years (Shanahan et al. 2011; Great Basin Bird Observatory 2012, 2013, 2014).

## Threats

An estimated 160,000 to 180,000 hectares of native riparian vegetation was estimated long the LCR in 1894 (Mearns 1907). By 1986, this native riparian vegetation had been reduced to roughly 25% (40,000 hectares) of its former extent (Anderson and Ohmart 1984; Younker and Anderson 1986). The halting of annual flooding, agricultural and urban development within historic flood plains, salt cedar 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 LCR's riparian system. Rosenberg et al. (1991) suggest that the resulting major losses of cottonwood-willow on the river were the initial and primary cause of yellow warbler declines. They also speculate that Sonoran yellow warblers nesting in replacement habitats of screwbean mesquite (*Prosopis pubescens*) and salt cedar may have experienced higher rates of breeding failure than in native habitats, thereby causing further declines of the species.

Cowbird (*Molothrus* spp.) parasitism poses a limited to moderate threat to yellow warblers. However, the current impact of cowbird parasitism remains unreported for Sonoran yellow warblers. East of the Sierra Nevada crest, 41% of 566 yellow warbler 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 salt cedar 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 2008).

The effects of predation on Sonoran yellow warbler productivity and population viability are unreported.

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# Sticky Buckwheat
# (*Eriogonum viscidulum* Howell)

## LEGAL STATUS

Sticky buckwheat (*Eriogonum viscidulum* Howell) are listed as a critically endangered species in Nevada, are on the Bureau of Land Management (BLM) sensitive species list, are listed on the Department of Conservation and Natural Resources, Nevada Natural Heritage Program's Sensitive List as imperiled, and are a covered species on two local conservation plans: the Lower Colorado Multiple-Species Conservation Plan (LCR MSCP) administered by the Bureau of Reclamation and the Clark County Multiple Species Habitat Conservation Plan administered by Clark County, Nevada.

## DISTRIBUTION

### Historical Range

The historic range of sticky buckwheat was restricted to the northeastern Mojave Desert, including the Muddy River and lower Virgin River watersheds (The Nature Conservancy 2007).

### Current Range

The known global distribution of sticky wild buckwheat is restricted and confined to Clark and Lincoln Counties, Nevada, and Mohave County, Arizona, with the majority of the current distribution of the species located in Clark County. They are endemic to the northeastern Mojave Desert Ecoregion (The Nature Conservancy 2007).

All known occurrences have been organized into 13 population groups centered on the confluence of the Muddy and Virgin Rivers. From north to south, these are:

- Two population groups north of the Virgin River drainage, one in Mohave County, Arizona (referred to as Arizona) and one in Lincoln County (referred to as Eastern Lincoln County) at the species northern extent

- Five population groups along the Virgin River drainage (referred to as Toquop Wash, Upper Virgin Valley, Lower Virgin Valley, Lower Virgin River, and Virgin River confluence)

- Two population groups along the Muddy River drainage (referred to as Upper Muddy River and Middle Muddy River) defining its northwestern distribution

- Two small population groups northeast and east of the Overton Arm of Lake Mead (referred to as Bitter Ridge and Lime Wash)

- Two population groups west of the Overton Arm of Lake Mead (referred to as Overton Arm and Black Mountains), the latter defining the species southernmost extent (The Nature Conservancy 2007).

## Populations Within the LCR MSCP Planning Area

Sticky buckwheat are present within the Lower Colorado River Multi-Species Conservation Program planning area, near the Overton Arm of Lake Mead (Powell 2003; Bangle 2005, 2010).

# LIFE HISTORY

## Description

Sticky buckwheat, a member of the buckwheat (Polygonaceae) family, are rare, sand-loving, annual plants endemic to Clark and Lincoln Counties in southern Nevada and Mohave County in northwestern Arizona. Sticky buckwheat can reach up to 40 centimeters tall with diffusely branched stems and a base rosette of leaves. They have small yellow flowers, and the stems and the branches are slightly sticky and are often covered with sand particles (Bureau of Reclamation 2004).

Sticky buckwheat plants have a lifespan of approximately 5 to 8 months from seed germination. Seeds germinate in winter following sufficient precipitation (about 15 to 25 millimeters in the northern Mojave Desert) (Beatley 1967). Their abundance varies annually and is dictated by temperature and moisture. There is increased germination and seed bank replenishment in years with above-average winter rains and when adequate temperature and moisture follows during the growing season. It is unknown how long seed banks remain viable (The Nature Conservancy 2007).

## Phenology

Sticky buckwheat plants flower from April through June (Nevada Heritage Program 2001b).

## Habitat

Sticky buckwheat are found in deep, loose, sandy soils in washes, flats, roadsides, steep Aeolian slopes, and stabilized dune areas at elevation of 1500 to 2500 feet (Department of Conservation and Natural Resources, Nevada Natural Heritage Program 2001a). They occur on slopes averaging about 8 degrees but can be on slopes as great as 53 degrees. They are found most often on west and east exposures (The Nature Conservancy 2007). The majority of sticky wild buckwheat populations are have been recorded on Triassic tufaceous sedimentary rocks, with some occasionally occurring on alluvial deposits. They are mostly mapped on Badland and Toquop fine sand soil associations but also occurs at a few scattered sandy sites in the Tertiary Muddy Creek Formation, and these locations often are capped by a caliche layer (Niles et al. 1995 *in* The Nature Conservancy 2007). Sands on which sticky wild buckwheat grow are delivered to their habitat primarily by fluvial transport (The Nature Conservancy 2007).

Sticky buckwheat are found growing in Mojave desert scrub with white bursage (*Ambrosia dumosa*), creosote bush (*Larrea tridentata*), big galleta grass (*Pleuraphis rigida*), littleleaf rattan (*Krameria parvifolia*), Indian rice grass (*Achnatherum hymenoides*), salt cedar (*Tamarix ramosissima*), arrowweed (*Tessaria sericea*), Geyer's milkvetch (*Astragalus geyeri* var. *triquetrus*), gravel milkvetch (*A. sabulonum*), desert trumpet (*Eriogonum trichopes*), Mormon tea (*Ephedra torreyana*), desert twin bugs (*Dicoria canescens*), breadroot (*Pediomelum* spp.), California croton (*Croton californicus*), sand dropseed (*Sporobolus cryptandrus*), and Fremont's dalea (*Psorothamnus fremontii*) (Department of Conservation and Natural Resources, Nevada Natural Heritage Program 2001a).

## Threats

Major threats to sticky buckwheat are competition from non-native Mediterranean/Arabian grass (*Schismus barbatus*), Russian thistle (*Salsola tragus*), salt cedar, Sahara mustard (*Brassica tournefortii*), and native (arrowweed) plants, and the potential rising of reservoir elevations. Some populations of sticky buckwheat can occur in open, bedrock sites below the high water line of Lake Mead where they established during years with low reservoir elevations (e.g., Lime Wash, Overton Arm, Black Mountains, and Virgin River Confluence sites) (The Nature Conservancy 2007). Additional threats are from development and off-highway vehicle use and, at select sites, trampling and grazing by trespass cattle and burros. Trampling by people is rare (Powell 1999, 2003; Bangle 2005, 2010). The species can withstand moderate temporary disturbance and is dependent on sand dunes or deep sand (Department of Conservation and Natural Resources, Nevada Natural Heritage Program 2001a).

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# Summer Tanager
(*Piranga rubra*)

## LEGAL STATUS

Summer tanagers (*Piranga rubra*) are not currently federally listed. In California, they are considered a species of special concern (breeding), Priority 1, and have been included on the list since inception (Remsen, Jr. 1978; California Department of Fish and Game 1992). Summer tanagers are also a covered species under the Clark County Multiple Species Habitat Conservation Plan.

## DISTRIBUTION

### Historic Range

Summer tanagers have been declining along edges of their range in most areas of the Eastern United States. They formerly bred in central Iowa, southern Wisconsin, throughout northern Illinois, and central Indiana (Robinson 1996). In the West, the species 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 the population decline in the lower Colorado River Valley (Hunter 1984); causes along the northern edge of the eastern range are unknown.

Cooper (1861), the earliest ornithologist to visit the lower Colorado River Valley, found summer tanagers to be "common," as did Grinnell (1914). Grinnell and Miller (1944) consider it to be "common within restricted range and habitat." They also describe the summer tanager's breeding range in California as the lower Colorado River (LCR) from the Nevada line south to the Mexican border. Specific records are recorded from Needles, California, north to the Nevada line; 25 miles below Ehrenberg, Arizona; and 8 miles below Picacho State Recreation Area, in California, downstream to Pilot Knob (Grinnell and Miller 1944).

### Current Range

Summer tanagers breed across the Southern United States and northern Mexico and winter from central mainland Mexico south to northern South America (Robinson 1996; American Ornithologists' Union [AOU] 1998). Two subspecies are currently recognized: (1) *P. r. cooperi* – breeds from southern California east to New Mexico and south in mainland Mexico to the States of Durango and Nuevo Leon and (2) *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 their attachment to mature riparian forests, these tanagers are localized over their western range, though “common” within this habitat in Arizona (Monson and Phillips 1981) (figure 1).

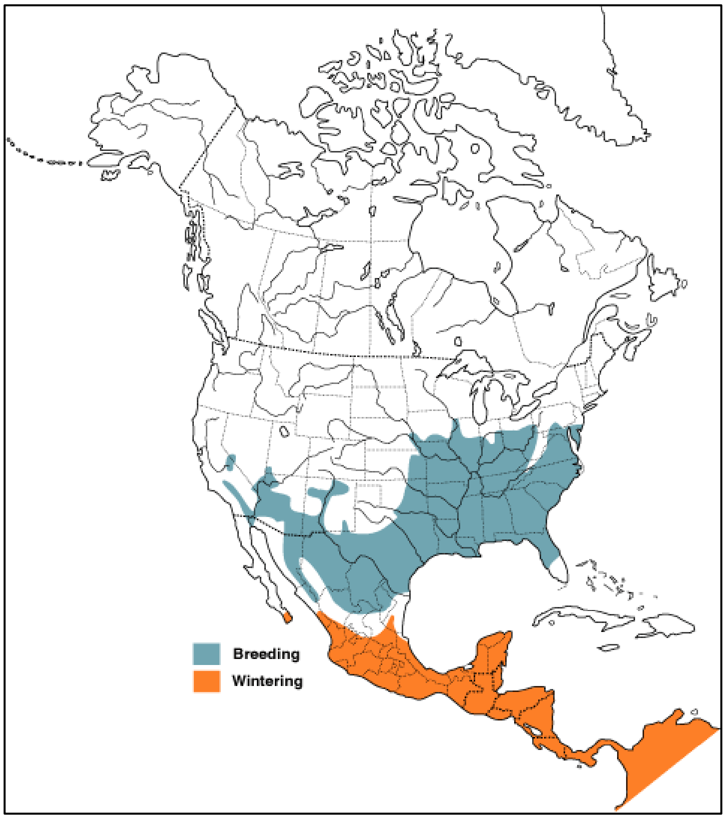

**Figure 1.—Distribution of summer tanagers in North and Middle America.**
This species winters south to northern South America (Robinson 1996).

## Populations Within the LCR MSCP Planning Area

By 1976, the number of summer tanagers had declined to an estimated 216 individuals along the LCR, well over one-half of these on the Arizona side, primarily in the Bill Williams River Delta (Rosenberg et al. 1991). Under the best of conditions, such as those found on a 20-hectare (ha) plot along the Bill Williams River (approximately 3 kilometers upstream of the Bill Williams River Delta) in 1977–78 (Anderson and Ohmart 1984), the estimated density of summer tanagers reached an average of 24 birds per 40 ha (range 20–30 birds per ha) in cottonwood-willow (*Populus* spp., *Salix* spp.) stands (Rosenberg et al. 1991). Hunter (1984) found 5 tanagers along the California side of the LCR; in Arizona, 5 were located in the large athel tamarisk (*Tamarix aphylla*) at the Havasu National Wildlife Refuge (lost to fire in 2008); 3 were located at an athel stand near Dome Valley, Arizona; up to 10 were located along the Bill Williams River Delta; and up to 7 were located at the Cibola National Wildlife Refuge (Cibola NWR). Hunter (1984) revised an estimate of 46 summer tanagers, which was based on the presence of available habitat, to no more than 10 pairs on the California side of the river after surveys were completed. The population continued to decline to an estimated 198 individuals in 1984 and 138 in 1986, reflecting habitat losses from prolonged flooding of the Bill Williams River Delta and the LCR (Rosenberg et al. 1991). Twenty-two males, with 3 on the California side of the river (S.A. Laymon and M. Halterman, personal communication *in* Rosenberg et al. 1991), were detected in 1986 during a comprehensive survey.

Between 1994 and 1996, Lynn and Averill (1996) conducted point count surveys at the four lower Colorado River national wildlife refuges. They reported 153 observations of summer tanagers across all refuges. McKernan and Braden (2002) summarized summer tanager observations within the lower Colorado River Valley and southern Nevada from 1996–2001. The species was present for at least 2 years at the sites in southern Nevada along the Virgin River, at the Pahranagat National Wildlife Refuge, at the Meadow Valley Wash, and in the lower Grand Canyon/upper Lake Mead area. In the lower Colorado River Valley, the species was present at Topock Marsh, Topock Gorge, Lake Havasu, Bill Williams River National Wildlife Refuge, Headgate Dam, Ehrenberg, Walker Lake, Draper Lake, Paradise Valley, Adobe Lake, and Taylor Lake. In the Picacho State Recreation Area, breeding tanagers were observed feeding at least one fledgling on July 19, 2000. Between 1999 and 2001, summer tanagers were observed irregularly in an 8-ha cottonwood revegetation site adjacent to the main campground at the Picacho State Recreation Area (Unitt 2008).

Based on surveys conducted system-wide along the LCR by the Great Basin Bird Observatory (GBBO) since 2007, summer tanagers “occur sporadically and in low numbers throughout the project area” (GBBO 2011). Data from area searches and spot mapping collected between April 2 and June 13, 2012, and analyzed using the double-sampling program. Bart and Hartley (2011) estimate a minimum population size of 199 summer tanager territories within the Lower Colorado River Multi-Species Conservation Program (LCR MSCP) planning area.

Table 1 includes data specifically from riparian habitat created under the LCR MSCP as well as from a restored site on the ‘Ahakhav Tribal Preserve (GBBO 2008a, 2008b, 2009, 2011, 2012, 2013). Further descriptions of these sites can be found under the “Technical Reports” tab at http://www.lcrmscp.gov.

The Bureau of Reclamation operates three Monitoring Avian Productivity and Survivorship stations along the LCR: (1) at the Cibola NWR Nature Trail, (2) at the Cibola Valley Conservation Area (CVCA), and (3) at the Beal Lake

Table 1.—Presence of summer tanager at LCR MSCP restoration sites and at the ‘Ahakhav Tribal Preserve (GBBO 2008a, 2008b, 2009, 2011, 2012, 2013)

| Site name and dates of planting | 2007* | 2008* | 2009* | 2010* | 2011* | 2012* |
|---|---|---|---|---|---|---|
| Beal (Havasu National Wildlife Refuge) (2002–12) | 0/0 | 0/P | 1/ | 1/ | 2/ | 1/ |
| ‘Ahakhav Tribal Preserve (2002–05) | 1/ | 1/ | 1/ | – | 0/ND | 1/ |
| Palo Verde Ecological Reserve (2006–12) | 0/P | 0/ND | 0/ND | 0/P | 0/ND | 0/ND |
| Cibola Valley Conservation Area (2006–10) | 0/P | 0/P | 0/ND | 0/ND | 2/ | 2/ |
| Cibola NWR Unit #1 (2002 – ongoing) | 0/P | 0/ND | 0/ND | 0/ND | 0/ND | 0/P |
| Yuma East Wetlands (2004 – ongoing) | – | – | – | – | – | 0/ |

* Number of territories detected/ND = if not detected, and P = if present but breeding not confirmed. Dashes indicate no survey was conducted.

Conservation Area (BLCA). Two summer tanagers each have been captured and color banded at the CVCA and BLCA, but the sample size of recaptures and re-sightings of these individuals is still too small to analyze for productivity and survivorship. One summer tanager at the BLCA was re-sighted in late September at the BLCA. In addition, recent data from the eBird database (www.eBird.org) indicate the presence of summer tanagers on the following dates at the Palo Verde Ecological Reserve: May 16 and July 18, 2011; June 17 and July 1, 2012; and May 31, 2013. Crews conducting surveys for yellow-billed cuckoos (*Coccyzus americanus*) (McNeil and Tracy 2013; McNeil et al. 2013; Parametrix, Inc., and Southern Sierra Research Station 2015) have detected summer tanagers incidentally at multiple sites along the LCR between Needles, California, and Yuma, Arizona, including the Havasu, Cibola, Bill Williams River, and Imperial National Wildlife Refuges, ‘Ahakhav Tribal Preserve, Palo Verde Ecological Reserve, Cibola Valley Conservation Area, and Picacho State Park (on the California side of the river).

The Bill Williams River National Wildlife Refuge continues to be a population center for this species, with the most breeding season reports within the lower Colorado River Valley (www.eBird.org). Other current locations where numerous sightings of summer tanagers occur during their breeding season include the Havasu National Wildlife Refuge (Pintail Slough, Bermuda Revegetation, Beal Lake Conservation Area, and Topock Marsh) and Rotary Park in Lake Havasu City (www.eBird.org). Other consistent sightings in the lower Colorado River Valley, likely in scattered cottonwood patches, occur at Mittry Lake, the Imperial National Wildlife Refuge, lower Gila River/Colorado River confluence, and Yuma West Wetlands (www.eBird.org).

In contrast to the precipitous decrease along the Colorado River, summer tanagers elsewhere in southern California have gradually colonized and spread. The species was first reported at Morongo Valley in 1962 and at 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 that 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 2008). 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 a little over 100 pairs (Unitt 2008).

# LIFE HISTORY

## General Description

Summer tanagers are large tanagers, about 17 centimeters long, with average masses of nearly 30 grams. They are most easily confused with congeneric scarlet (*P. olivacea*) and hepatic (*P. flava*) tanagers. Adult males are distinguished from scarlet tanagers by their paler plumage, with more rose or orange-red than intense scarlet, and red, rather than black, wings and tail. The adult, male hepatic tanager has a dusky gray ear patch and an even duller red plumage that often appears gray. Female summer tanagers are usually brownish or orange-yellow, lack a greenish cast to their plumage, and have narrow, yet conspicuous, yellowish edging on the wing coverts, which makes the wings appear to have same color as the body. Some older, female summer tanagers apparently become partly or even completely pigmented as males, with plumage ranging from yellow to orangish red to red. Some females even show a patchy plumage with several shades of red scattered across the body. The typical immature, male summer tanager has a distinctive spotted or splotched plumage with orangish red and red patches on an otherwise yellowish plumage. Many immature tanagers 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's call is a distinctive "pit-i-tuck," with more staccato than the western tanager and very different from the calls of scarlet ("chip-brrr") and hepatic (soft "chuk") tanagers (Robinson 1996).

## Breeding

Summer tanagers are rare to uncommon summer breeders along the LCR, arriving from mid-April to early May and departing usually in early October; their breeding season extends from mid-May through July. Males arrive first and aggressively establish territories by the end of April and the beginning of May. Nest building and courtship feeding take place in mid-May, and first eggs are hatched by June 1. First broods fledge in mid- or late June, with most pairs re-nesting with second broods in late July. Rosenberg (1991) found four nests in cottonwood trees and three nests in willow trees, between 8–15 meters high.

## Feeding

Summer tanagers forage primarily for large insects, moving 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) along the LCR 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

Summer tanagers prefer structurally well-developed cottonwood-willow stands where they may attain densities of 20–30 birds per 40 ha (Rosenberg et al. 1991). In Arizona, summer tanagers have bred in stands of athel tamarisk and, at higher elevations, honey mesquite (*Prosopis glandulosa*) and salt cedar (*Tamarix* spp.). Rosenberg et al. (1991) suggest that tree height (at least 9 meters) and canopy closure are the critical variables making the habitat suitable for summer tanagers. They noted that the species nests in salt cedar and mesquite (*Prosopis* spp.) 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 density along perennial drainages where continuous woodlands of large Fremont cottonwood (*Populus fremontii*) and Goodding's willow (*Salix gooddingii*) exist. Clearly, tall, shady trees are the most critical element (Corman 2005).

Along the south fork of the Kern River, T. Gallion (in Robinson 1996) found summer tanagers using areas of 9 to 11 ha. Along the Colorado River, Rosenberg et al. (1991) record a density of 20–30 birds per 40 ha of suitable habitat.

## Threats

Removal of riparian forests is the most direct threat to summer tanagers in California. In addition, habitat degradation, through fragmentation and the lowering of water tables, compound the effects of clearing, cutting, and burning of trees. If the heat-moderating qualities of the leafy cottonwoods and willows are critical to the nesting success of summer tanagers and other desert birds nesting in midsummer, as implied by Rosenberg et al. (1991), fragmentation of a once continuous forest could reduce its ability to create a cooler microclimate. Temperatures, even in the shade of remaining scattered cottonwoods, could rise above the 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, though they visited it occasionally.

Unnatural water regimes, in combination with the invasion of salt cedar, are also a threat. Floods in 1983, 1984, and 1986 killed most remaining cottonwoods along the LCR, and high soil salinity, prolonged inundation, and fire favored their replacement by salt cedar (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 salt cedar, proliferation of other exotic plants, such as giant reed (*Arundo donax*) and Russian olive (*Elaeagnus angustifolius*), displaces suitable summer tanager habitat.

Fire is a serious threat to summer tanager habitat. Burning of riparian forests along the Colorado River favors salt cedar 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 more mesic coastal slope (Unitt 2008).

Cowbird (*Molothrus* spp.) parasitism has not been identified as a serious threat to summer tanagers 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 brown-headed cowbirds (*Molothrus ater*) (T. Gallion *in* Robinson 1996). Brown-headed cowbirds were "common" along the Colorado River in 1910 (Rosenberg et al. 1991), and they may pose a population-level threat only when tanager numbers are already greatly reduced by habitat loss and degradation. Larger bronzed cowbirds (*Molothrus aeneus*) may pose more of a threat to summer tanagers than smaller brown-headed cowbirds. Bent (1958) calls the summer tanager a "fairly regular" victim of bronzed cowbirds in Sonora; therefore, the increased population of bronzed cowbirds in California may raise concern (Unitt 2008).

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# Threecorner Milkvetch (*Astragalus geyeri* var. *triquetrus* [Gray] Jones)

## LEGAL STATUS

Threecorner milkvetch (*Astragalus geyeri* var. *triquetrus* [Gray] Jones) were previously listed as a Federal Category 2 candidate under the Endangered Species Act. Currently, they are listed as a species of concern with the U.S. Fish and Wildlife Service. In Nevada, they are listed under Nevada Revised Statute (NRS 527.260) as critically endangered and are a special status species with the Bureau of Land Management.

## DISTRIBUTION

### Historical Range

Historically, threecorner milkvetch were found in southeastern Nevada and northwestern Arizona (Powell 1998).

### Current Range

In Nevada, threecorner milkvetch are found in Clark County along the Muddy River, the lower Virgin River, and Lake Mead (Overton Arm, Virgin Basin, and Lower Basin) (Department of Conservation and Natural Resources, Nevada Natural Heritage Program 2001). In Arizona, they are currently located in Mohave County in Sand Hollow Wash, Horsethief Canyon, and Beaver Dam Wash (Arizona Game and Fish Department 2005).

### Populations Within the LCR MSCP Planning Area

Threecorner milkvetch occur within the Lower Colorado River Multi-Species Conservation Program planning area, along Lake Mead at Middle Point and Ebony Cove (Virgin Basin), and Sandy Cove (Lower Basin) (Powell 1999, 2003; Bangle 2005, 2010).

# LIFE HISTORY

## Description

Threecorner milkvetch are members of the legume (Asteraceae) family. They are slender, ashy-pubescent herbs, with stems 10–20 centimeters (cm) long and compound leaves 3–5 cm long, with approximately nine elliptical, 4–15 millimeter (mm) long leaflets. Each raceme has to two to eight 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. The pods are 1 cm long, oblong, curved, and triangular in cross-section, with a groove on the lower side (Arizona Game and Fish Department 2005).

## Phenology

Threecorner milkvetch are winter annuals with a lifespan of 5 to 8 months from germination to plant death (The Nature Conservancy 2007). They flower in April and May, with fruit setting in 4–6 weeks after flowering (Arizona Game and Fish Department 2005).

## Habitat

Threecorner milkvetch are found on areas with stabilized sand, frequently with sparse gravel (Arizona Game and Fish Department 2005). Their locations are closely related with the Muddy Creek Formation, a Tertiary-aged sedimentary rock widely exposed along Lake Mead’s portion of the Colorado River and its tributary valleys (The Nature Conservancy 2007). They have been recorded within the elevation range of 1200 to 2450 feet) (The Nature Conservancy 2007). They typically occur on areas with less than 2-degree slopes, but they can be found on slopes as great as 21 degrees. Threecorner milkvetch occur on all exposures; west and southwest exposures are the most common, and east and northeast the least common. This pattern suggests that pods and seeds are likely wind transported (The Nature Conservancy 2007).

## Threats

Major threats to threecorner milkvetch are off-road vehicle use and invasion of non-native plants (Sahara mustard [*Brassica tournefortii*], Mediterranean grass [*Schismus barbatus*], and Russian thistle [*Salsola tragus*]). Trampling by boaters along the Lake Mead shoreline is considered a minor threat. Fluctuations in reservoir elevation are a potential threat for some populations (Bangle 2005, 2010).

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# Vermilion Flycatcher (*Pyrocephalus rubinus*)

## LEGAL STATUS

Vermilion flycatchers (*Pyrocephalus rubinus*) are not currently federally listed nor do they have any additional legal status in Arizona, Nevada, or California. Flycatchers are 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 population in California, they are vulnerable to extirpation should a threat materialize.

## DISTRIBUTION

Comprising 12 subspecies, vermilion flycatchers range from the Southwestern United States south to central Argentina and Uruguay (figure 1) (American Ornithologists' Union [AOU] 1998; Wolf and Jones 2000). The subspecies found within the Lower Colorado River Multi-Species Conservation Program (LCR MSCP) planning area 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) consider vermilion flycatchers 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 meters in elevation. In 1947, at least three pairs of vermilion flycatchers were reported nesting in the upper Mohave River drainage approximately

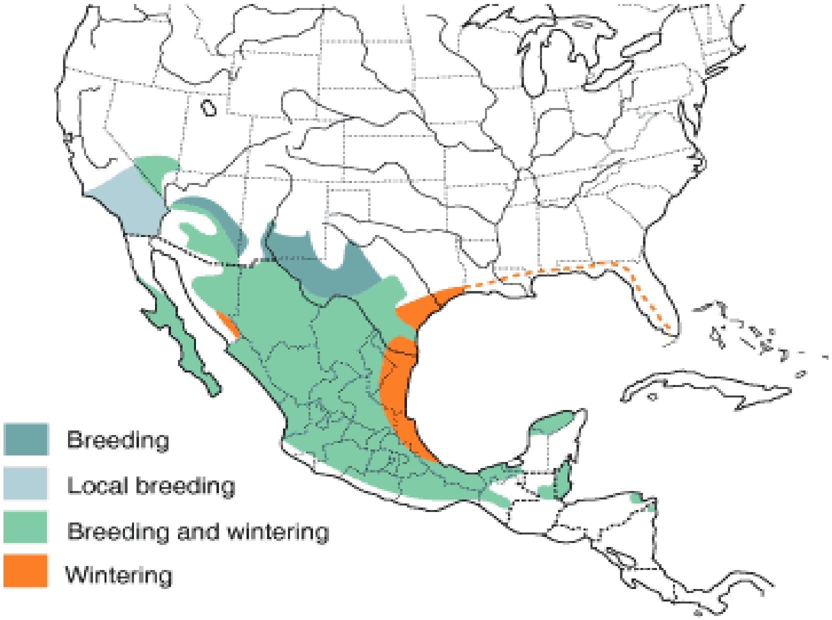

**Figure 1.—Distribution of vermilion flycatchers.** This species winters north and east locally to the dashed orange line (Wolf and Jones 2000).

100 miles from other known breeding sites on the lower Colorado River (LCR) or the Salton Sea (Jaeger 1947). 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); the populations also have declined in the Imperial Valley, where they are now considered “rare” breeders (Patten et al. 2003).

Vermilion flycatchers have undergone a significant range shift in California during the past five decades. This change in status, from breeders to early winter visitors, has been documented in other parts of the flycatcher’s Sonoran Desert range (Rea 1983). Vermilion flycatchers are now virtually unknown as breeders in the Sonoran Desert of California (Rosenberg et al. 1991), but breed in many locations in the Mojave Desert, almost all of which are well above 150 meters in elevation.

Serena (1981) listed locations where vermilion flycatchers were found in California in the late 1970s and1980s. These areas included the Blythe Golf Course and Clark Ranch. There was an unmated female at Clark Ranch during the summer of 1982. Hunter (1984) reports 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 main stem at the Parker Dam residences on April 21. This pair fledged two young by June 9. No other individuals were found during this survey.

During 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 (*Prosopis* spp.), willows (*Salix* spp.), and cottonwoods (*Populus* spp.) in southern and central Arizona but rather local along the Salt and Colorado River Valleys (Phillips et al.1964).

## Current Range

The northern limit for vermilion flycatchers is southern Nevada, with the most significant breeding population in Clark County, in the Warm Springs Natural Area of the Moapa Valley. Sullivan and Titus (2001) estimate 25 vermilion flycatchers in this area; however, a fire that occurred on Warm Springs Ranch in 2010 may have affected their status there. The species is occasionally reported further north at Pahranagat and Ash Meadows National Wildlife Refuges. Sightings as far north as Fallon, Nevada, are considered accidentals (Alcorn 1988).

Vermilion flycatchers breed in Arizona from the northwest and Mogollon Rim, south throughout the State. Flycatchers are 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. The species was also found regularly to west-central Arizona along the Hassayampa, Bill Williams, Santa Maria, and Big Sandy River systems. Observers working on the Arizona Breeding Bird Atlas found vermilion flycatchers to be a local breeder along the LCR (Averill-Murray and Corman 2005). Flycatchers are reported as rare and local breeders along the lower Verde, Salt, and Colorado Rivers (Monson and Phillips 1981; Rosenberg et al. 1991). They also breed 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). Vermilion flycatchers breed in western and central Texas and occasionally in central and western Oklahoma (Wolf and Jones 2000).

The breeding range of vermilion flycatchers has declined throughout southern California (Rosenberg et al. 1991). They are currently 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 residents throughout all but the northernmost portion of their breeding range in the United States, Mexico, and Central America. This species winters outside of their 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).  Their winter range fluctuates with weather conditions.  In some winters, flycatchers will wander along river corridors outside their normal range (Grinnell and Miller 1944).  Vermilion flycatchers winter where the average minimum January temperature is usually above -1 degree Celsius.  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 the LCR MSCP Planning Area

Data on the vermilion flycatcher population along the LCR prior to 1992 comes mainly from Rosenberg et al. (1991), collected from 1974–84 as part of the "Lower Colorado River Bird Study."  At that time, breeding populations occurred at the Bill Williams River National Wildlife Refuge (Bill Williams River NWR); with a few scattered pairs at the Blythe Golf Course (one pair), Clark Ranch (one pair); near Poston, Arizona (two pairs); the Parker Dam residences and Willow Valley Estates (one pair); near Yuma, Arizona (two pairs); and the Bill Williams River Delta (four to six pairs) (Serena 1981; Rosenberg et al. 1991).

The vermilion flycatcher population on the Bill Williams River NWR seems to have increased after the LCR bird study was officially completed in 1984, and they remained fairly common there throughout the 1990s, particularly at the eastern portion of the refuge (B. Raulston, personal observation; K. Blair 2013 personal communication; B. Raulston 2013).  Neale et al. 1994, working with Michael Morrison, University of Arizona, documented 10 nests in April and May 1993 from Mineral Wash to Planet Ranch (B. Sacks 2013, personal communication).  In addition to the 10 males associated with these nests, they also noted 8 single males and 3 paired (not yet nesting) males during that period of time.

In 1994, Annalaura Averill and Suellen Lynn, also under the direction of Michael Morrison, determined the relative abundances of riparian birds, including vermilion flycatchers, using a variable circular-plot method to conduct point counts surveys on the four lower Colorado River national wildlife refuges (Imperial, Cibola, Bill Williams River, and Havasu) (Lynn and Averill 1996). Although vermilion flycatchers were not detected elsewhere along the LCR during this time, L. Averill (unpublished) documented at least 30 pairs breeding on the Bill Williams River NWR and at least 62 nesting attempts (L. Averill-Murray, 2013, personal communication). Although no birds were banded during these studies, vermilion flycatchers are very territorial, and with minimal observation, individual pairs are easier to distinguish than many other species (Rosenberg et al. 1991; B. Raulston 2013, personal observation).

During focused surveys for southwestern willow flycatchers (*Empidonax traillii extimus*)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). Since 1998, data from strip transects surveyed by Kathleen Blair, Bill Williams River NWR, and various volunteers documented a steady decline in observations of vermilion flycatchers on the Bill Williams River NWR (figures 2 and 3). Christmas Bird Counts along the LCR (figure 4) indicate vermilion flycatchers are present year round, but the relationship between wintering and breeding populations is unknown, as banding has not been conducted.

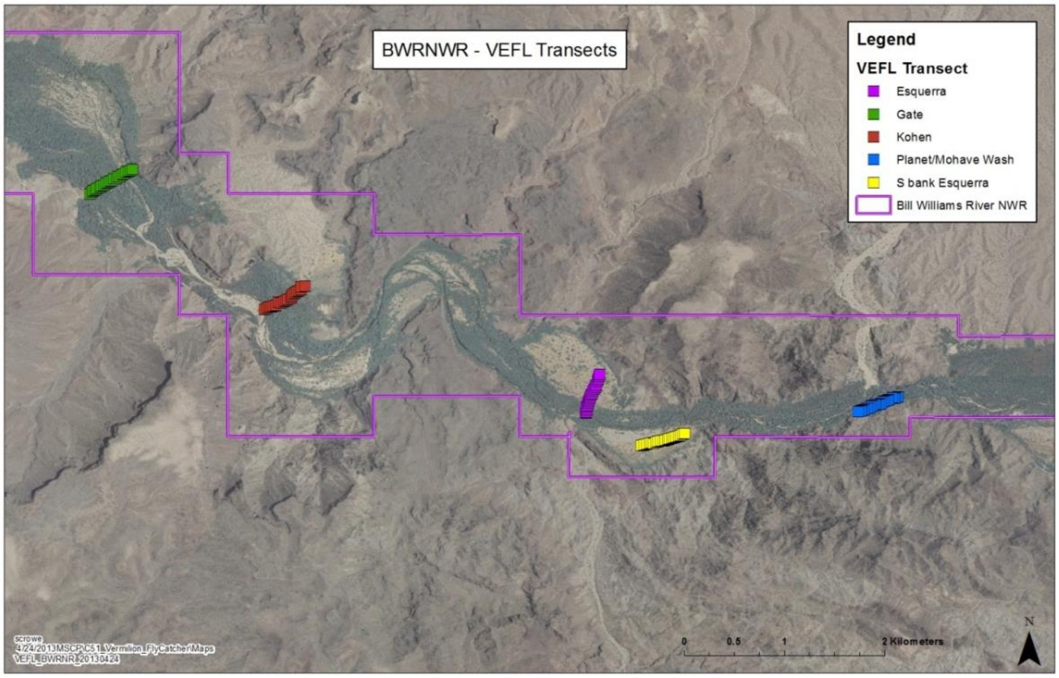

**Figure 2.—Locations of transects on the Bill Williams River NWR.**

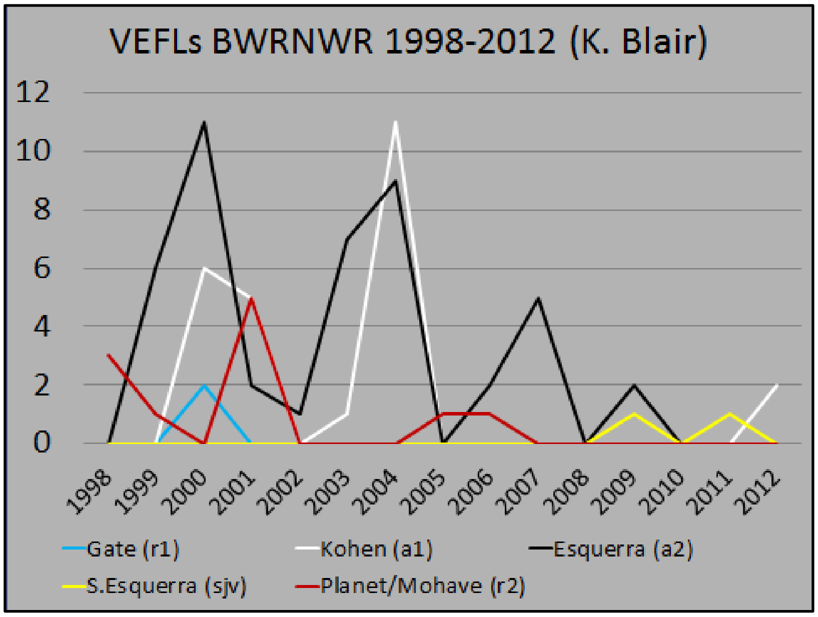

**Figure 3.—Vermilion flycatchers detected on the Bill Williams River NWR since 1998.**

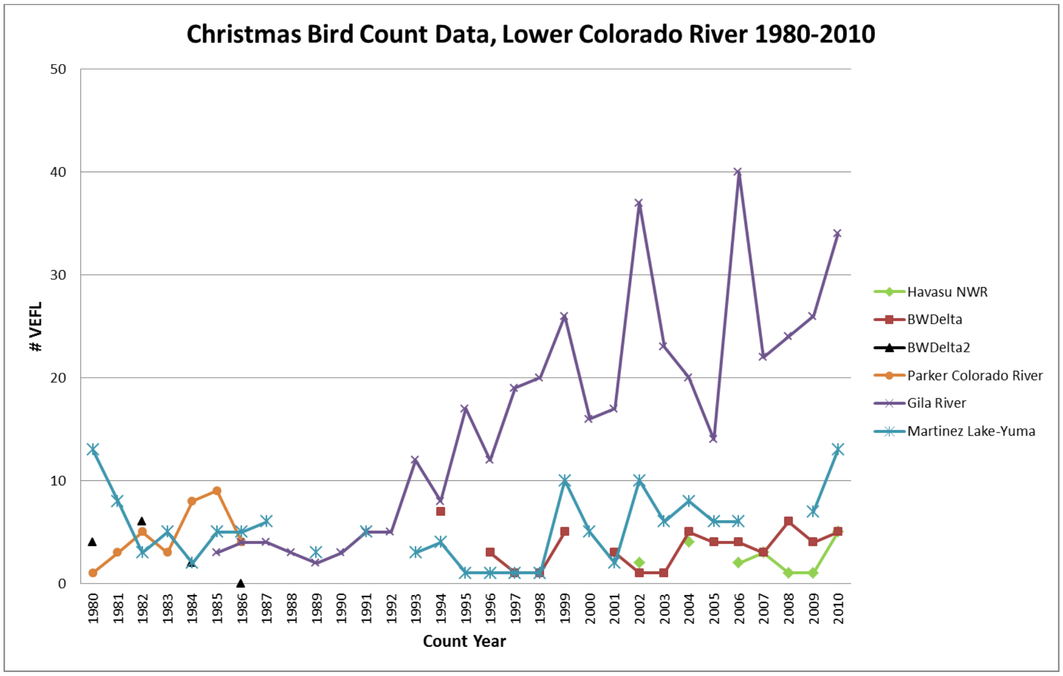

**Figure 4.—Christmas Bird Count results, LCR, 1980–2010.**

Vermilion flycatchers are still very localized along the LCR; they are not present in all, or even most, of the cottonwood-willow type I–IV or mesquite type III where available. The species has declined even on the Bill Williams River NWR, where this habitat is still present in abundance (see figure 3). Vermilion flycatchers remaining along the LCR are easily found and there are multiple records in the eBird database. Most of these sightings occur in irrigated grassy parks and golf courses rather than native habitats.

Results from surveys conducted throughout the LCR by the Great Basin Bird Observatory (GBBO) (2008, 2009, 2010, 2011, 2012, 2013) indicate that the number of vermilion flycatchers found was too small to conduct an analysis of habitat.

There are several locations along the LCR where bird projects are occurring under the LCR MSCP. When vermilion flycatchers are observed incidentally at these locations, they are reported. During 2012, a total of 40 locations along the LCR from Yuma, Arizona, to Needles, California, which are not included in other projects, were visited by LCR MSCP staff between February 2 and April, 19, 2012. Information from grounds workers, employees, residents, or the general public was used to select areas for followup searches. Vermilion flycatchers were present at eight locations between Yuma and Lake Havasu City, Arizona, and nesting was documented at four sites. The locations where vermilion flycatchers were present included the Blythe Cemetery, River Lodge Golf Course (near Parker, Arizona), Big River Community Park (on the California side of LCR, near Parker, Arizona), Emerald Canyon Golf Course (Parker, Arizona), La Paz County Park (Parker, Arizona), and the Hidden Shores Resort (north of Yuma, Arizona). In addition, vermilion flycatchers were reliably reported by others in 2012 from Rotary Park (Lake Havasu City, Arizona), Quartermaster Depot State Park (Yuma, Arizona), ‘Ahakhav Tribal Preserve (Parker, Arizona), and McIntyre Park (Blythe, California). Breeding was confirmed at River Lodge Golf Course, Big River Community Park, La Paz County Park, and the Hidden Shores Resort.

This species is occasionally found in areas restored with native cottonwood and willow. A single male vermilion flycatcher was detected wintering at Cibola National Wildlife Refuge from 2002 to 2004 (Bureau of Reclamation, unpublished data) in the Cibola National Wildlife Refuge Nature Trail restoration site. There are several nesting pairs in the park at the ‘Ahakhav Tribal Preserve, which consists of irrigated grass with scattered mesquites. In January 2012, a male vermilion flycatcher was seen at the Palo Verde Ecological Reserve (B. Anderson, 2013, personal communication), and another individual was observed at the Cibola Valley Conservation Area in a large, remnant cottonwood tree on the edge of the created cottonwood-willow habitat adjacent to the agricultural fields.

# LIFE HISTORY

## General Description

A vermilion flycatcher is a small flycatcher with a length of about 13–14 centimeters and a mass of 11–14 grams. 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 the wings and tail, colored blackish brown. The adult female has the top of the head, ear coverts, and remaining upperparts, including the 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 plumage is similar throughout the year. The immature female is similar to the adult female, except the posterior underparts are yellowish. The immature male shows delayed plumage maturation, remaining similar to the adult female throughout summer of the second calendar year. The underparts of the immature male are extensively covered with salmon pink or pale orange-red (Wolf and Jones 2000).

## Breeding

Despite the vermilion flycatcher's conspicuousness, their biology remains poorly known (Wolf and Jones 2000). They are mainly summer visitors to the Southwestern United States, although they regularly winter in the Sonoran and Chihuahuan Deserts and in cismontane southern California. Breeding birds of the Colorado Desert are generally residents, 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). Their 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. Observers working on the Arizona Breeding Bird Atlas confirmed breeding (nests with young) through early August, with a peak in activity between late April and late June (Averill-Murray and Corman 2005).

Vermilion flycatchers are monogamous. The nest site is chosen based on the nest site showing display of males. They fly around to potential nesting sites and gives a soliciting call to the nearby females, encouraging them to take a look. The males 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 females is usually within 200 meters of the male's preferred nest site. Females will often ignore displaying males, 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. Males will retreat when they observe females 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 they are lined with down, feathers, and hair. The 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 two to three oval-shaped eggs, and 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, females begin 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 males feed 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. The 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 one-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 May 20 to June 10 (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 meters of the ground, with the least amount of foraging occurring over water. Vermilion flycatchers are sit-and-wait predators, 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

During their 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, willows, oaks (*Quercus* spp.), mesquites, and sycamores (*Platanus* spp.) line streams (Wolf and Jones 2000). When vermilion flycatchers formerly bred in the Sonoran Desert of California, they were associated with low-lying, open riparian areas with accessible water (either pooled or flowing) and dominated by mesquite, willow, and Fremont cottonwood (*Populus fremontii*) (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).

Along the LCR, vermilion flycatchers are most often found in riparian woodlands 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 (*Salix gooddingii*), Fremont cottonwoods, mesquites, Arizona walnuts (*Juglans major),* Arizona sycamores, (*Platanus wrightii*), desert willows (*Chilopsis linearis*), *Acacia* spp., and Palo verde (*Parkinsonia* spp.) but sometimes in non-native trees such as elms (*Ulmus* spp.), olives (*Olea europaea*), black locusts (*Robinia pseudoacacia*), salt cedar *(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). Observers working on the Arizona Breeding Bird Atlas 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 meters above ground (Rosenberg et al. 1991; Wolf and Jones 2000).

In central Arizona and northern Mexico, vermilion flycatchers were not found in areas where Fremont cottonwoods form dense canopy and mesquite form thick understory. Flycatchers breed 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 (relative frequency > 10%), Arizona ash (*Fraxinus velutina* (relative frequency < 3%), and rarely Arizona sycamore (*Platanus wrightii*) and Mexican elderberry (*Sambucus mexicanus*). Scattered scrubs include salt cedar (*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%.

As described in Carothers (1974), flycatchers also breed in mesquite bosques, where honey mesquite is the dominant tree species, along with desert willow (*Chilopsis linearis*), Goodding's willow, Fremont cottonwood, salt cedar, and blue Palo verde (*Cercidium floridum*). The scrub community was composed of honey mesquite, catclaw (*Acacia greggii*), seep-willow, wait-a-minute (*Mimosa buincifera*), wolfberry (*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. Flows from warm springs located on Moapa Valley National Wildlife Refuge, the Warm Springs Natural Area, and the Warm Springs Recreational Ranch in southern Nevada converge in tributaries that form the headwaters of the Muddy River, which flows into Lake Mead (Southern Nevada Water Authority 2011). Surrounded by Mojave desert scrub, the Moapa Valley accommodates the flycatcher's need for a mosaic of mesquites and riparian forests for nesting and flooded fields for catching flying insects (Patten 2006).

## Threats

The primary threat to vermilion flycatchers along the LCR is loss of riparian woodlands. The destruction of much of the native riparian habitat along the LCR, and its replacement in many areas by non-native salt cedar, 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 manmade 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 of vermilion flycatchers in California, but they appear to be uncommon hosts (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 brown-headed cowbirds.

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# Western Red Bat
# (*Lasiurus Blossevillii*)

## LEGAL STATUS

Western red bats (*Lasiurus blossevillii)* are not federally listed as threatened or endangered. They were 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, western red bats are a Nevada species of conservation priority and are protected and considered sensitive (Nevada Department of Wildlife 2005). In California, they are proposed as a mammal of special concern (Bolster 2005). The U.S. Forest Service considers western red bats sensitive animals in Region 3 (AGFD 2003). The Western Bat Working Group (1998) lists them as a species of “Red or High” priority, the highest priority available.

## DISTRIBUTION

### Historical and Current Range

The historical range of western red bats is believed to mirror their current range, where available habitat occurs, including western, central, and southern Nevada. The entire States of California and Arizona are considered within their range. One range map contains all of Washington, Oregon, Utah, Colorado, Wyoming, Idaho, Montana, New Mexico, and most of western Texas, though there are no known records of western red bats in Washington, Oregon, Idaho, or possibly Wyoming (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 red bats were once thought to range as far north as British Columbia due to a single specimen, but a recent genetic evaluation determined that the specimen, as well as two recent red bats found at a wind energy facility in northeastern British Columbia, were all eastern red bats (*Lasiurus borealis*) (Nagorsen and Paterson 2012). Western Texas is also an area of potential overlap between western and eastern red bats, 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 the LCR MSCP Planning Area

Western red bat demographics within the Lower Colorado River Multi-Species Conservation Program (LCR MSCP) planning area are not well known. Historically, no red bats had been reported along the lower Colorado River (LCR). The closest record along the LCR was of three female red bats collected in July 1902 on Big Sandy Creek (50 miles east of Topock, Arizona) (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 Biological Consulting 2006). In January 2002, a male red bat was mist netted on the Bill Williams National Wildlife Refuge approximately 7 miles from Lake Havasu (Brown 2006). During a survey from 2001 to 2002, red bats were recorded acoustically at the Bill Williams River National Wildlife Refuge, Havasu National Wildlife Refuge, and the Imperial National Wildlife Refuge, although they only accounted for 0.14 percent of the total call minutes (Brown 2006). The Havasu National Wildlife Refuge is located along approximately 30 miles of the Colorado River from Needles, California, to Lake Havasu City, Arizona. The Imperial National Wildlife Refuge is located along about 30 miles of the Colorado River 38 miles north of Yuma, Arizona. Prior to LCR MSCP habitat creation efforts, red bats were rarely detected acoustically, and an individual had never been observed on the main stem LCR. In February 2009, the first western red bat was captured at the 'Ahakhav Tribal Preserve within a habitat restoration site that was funded by the Bureau of Reclamation (Calvert and Neiswenter 2012). This was following winter acoustic monitoring that occurred a few weeks prior in which a high number of calls attributable to red bats were recorded (Broderick 2012a). Since that time, western red bats have now been captured at four additional habitat creation areas. At the Palo Verde Ecological Reserve and Cibola Valley Conservation Area, red bats have been captured during both winter and summer surveys, and both reproductive males and females have been captured. At Cibola National Wildlife Refuge Unit #1, reproductively active males have been captured in late summer. One red bat was also captured at Yuma East Wetlands in May 2012 (Calvert 2013; unpublished data).

# LIFE HISTORY

## General Description

Western red bats were previously recognized as a subspecies of the eastern red bat (*Lasiurus borealis teliotis*) (Cockrum 1960). They were 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. The wings are long, narrow, and pointed. It has a distinct bib under the neck, which contrasts greatly with the black wing membrane. The ears are 11–13 millimeters in length, low and rounded, and the tragus is short and blunt. The male is usually more colorful than the female. The forearms measure 38–43 millimeters, they weigh 7–15 grams, and the wingspan is 29.0–33.2 centimeters. Western red bats differ only slightly from eastern red bats, which are generally larger and have more frosted hairs. When possible, it is best to distinguish the two species by range. Western red bats can be distinguished from other bats not in the genera *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 western red bats are hoary bats (*L. cinereus*) and western yellow bats (*L. xanthinus*). Hoary bats are larger (forearm 5.0–5.4 centimeters) and have black fur around the edges of the ears. Western yellow bats are also larger (forearm 4.5–5.0 centimeters), 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. Females with young may roost in small maternity groups (Pierson et al. 2011; unpublished data). They normally migrate south in winter and may be active in areas with temperatures as low as 12–18 degrees Celsius (55–65 degrees Fahrenheit). Red bats active in winter have been found along the LCR (Calvert and Neiswenter 2012).

## 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. The estimated time for 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 600–1,000 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 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 meters away and, on average, will attack prey every 30 seconds. Western red bats are found to be successful 40 percent of the time (AGFD 2003).

# Habitat

Like other members of the genus *Lasiurus*, western red bats primarily roost in trees (Shump, Jr. and Shump 1982; Cryan 2003; AGFD 2003). Specifically, in the Southwestern United States, they are found in desert riparian areas. Tree species in these areas may include, but are not limited to, Fremont cottonwoods (*Populus fremontii*), Goodding's willows (*Salix gooddingii*), and Sycamores (*Platanus* spp.). Although western red bats may roost in any of these, they primarily roost in cottonwoods (*Populus* spp.). 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). During a roosting study along the LCR, most red bat roosts were in cottonwoods, and preliminary data indicate western red bat roosts are better predicted based on patch scale characteristics versus the individual tree scale, but they do appear to prefer trees with a larger crown width (Mixan et al. 2014). Red bats have been observed occasionally roosting in cave-like situations and in the boot of the saguaro cactus (*Carnegiea gigantea*), a hardened area of scar tissue that forms a hollow shape inside the cactus from cavity excavation by woodpeckers (E.L. Cockrum 1992, personal communication *in* AGFD 2003). Palo verde (*Cercidium floridum*) trees have been used as roost sites for the sympatric hoary bat (Brown 2006).

The elevation of these habitats may vary from 1900 to 7200 feet. Roost sites range from a few feet off the ground to more than 40 feet high. Red bats prefer heavily shaded areas, which are open underneath, enabling them to drop into flight. Williams (2001) found that the species will use a variety of habitats 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 four habitat types, with the riparian marsh being the least used.

An acoustic study that compared cottonwood-willow, mesquite (*Prosopis* spp.), salt cedar (*Tamarix* spp.), and marsh habitat types found that western red bat occupancy was much higher in cottonwood-willow habitat compared to all other habitat types (Vizcarra et al. 2010). A more recent acoustic study of large-scale (more than 50 acres) habitat creation areas along the LCR found that red bats responded quickly to newly planted habitat (dominated by cottonwood and willow [*Salix* spp.] trees). Within 3 years, western red bats showed a significant increase in acoustic activity at these young habitat creation areas, with no increase in adjacent habitat that consisted of agricultural fields and stands of non-native salt cedar (Broderick 2012b).

## Threats

Predators known to be threats to red bats include birds of prey, roadrunners, opossums (*Didelphis virginiana*), and domestic cats (Shump, Jr. 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 (Picidae) and raccoons (*Procyon lotor*) 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 western red bats in those areas. These forests may be important to not only resident western red bats but also to migrants (AGFD 2003). Eastern red bats have been observed hibernation roosting in leaf litter on the forest floor, where fire can be a threat (Moorman et al. 1999).

Human-caused threats include barbed wire 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 some people have about bats has caused them to harass or even harm bats. Wind energy facilities have become a more recent threat to western red bats, as the bats may collide with wind turbine blades or experience barotrauma (a condition in which a rapid air pressure reduction near rotating blades of the turbine causes tissue damage in the lungs due to the rapid expansion of air in the lungs that does not accommodate exhalation) (Baerwald et al. 2008). Migrating species are mostly affected by wind energy facilities, with the vast majority of fatalities occurring in late summer or fall. Most of these species are tree-roosting bats, including both eastern and western red bats (Kunz et al. 2007). Some theorize that tree-roosting bats, which mate during this time period, are looking for the tallest trees in the landscape to find a mate, and wind energy facilities may be providing artificial stimuli that attract them (Cryan 2008; Cryan and Barclay 2009).

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# Western Yellow Bat (*Lasiurus Xanthinus*)

## LEGAL STATUS

Western yellow bats (*Lasiurus xanthinus*) are not federally listed as threatened or endangered. They were 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, western yellow bats are a Nevada Species of Conservation Priority (Nevada Department of Wildlife 2005). The California Department of Fish and Game (now the California Department of Fish and Wildlife) has proposed them as a species of special concern (B. Bolster 2005, personal communication). The Western Bat Working Group (1998) lists western yellow bats as a species of "Red or High" priority, the highest priority available.

## DISTRIBUTION

### Historical Range

The first known occurrence of western yellow bats in the United States was 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 the 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 and 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 and Wilson 2002).

### Current Range

General range maps for western yellow bats 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 western and central Mexico (Kays and Wilson 2002; NatureServe 2005). 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 western yellow bats have expanded their range across the Southwestern United States northward with the introduction of the Washington fan palm (*Washingtonia robusta*) (AGFD 2003). They have also been confirmed farther east, in Big Bend National Park in Texas (Higginbotham et al. 1999).

## Populations Within the LCR MSCP Planning Area

Western yellow bat populations are not well known within the Lower Colorado River Multi-Species Conservation Program (LCR MSCP) planning area. 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 detected acoustically at the Bill Williams River National Wildlife Refuge, Imperial National Wildlife Refuge, and north of Parker, Arizona (Brown 2006). Western yellow bats have been detected acoustically during all months, except for January, at the Las Vegas Wash (O’Farrell Biological Consulting 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 planning area.

Surveys have been used to detect yellow bats in most sections along the LCR (Vizcarra et al. 2010). Acoustic surveys within riparian restoration areas along the LCR have showed increasing activity levels of yellow bats as the vegetation matured (Broderick 2012). Capture surveys have confirmed yellow bats at seven riparian restoration areas, including the ‘Ahakhav Tribal Preserve near Parker, Arizona; Palo Verde Ecological Reserve near Blythe, California; Cibola Valley Conservation Area and Cibola National Wildlife Refuge Unit #1 near Cibola, Arizona; Imperial National Wildlife Refuge; Pratt Restoration Site near Laguna Dam; and Yuma East Wetlands near Yuma, Arizona. Reproductively active yellow bats have been confirmed at most of these sites, indicating a breeding population along the LCR (Calvert 2010, 2013; Vizcarra et al. 2010).

# LIFE HISTORY

## General Description

Western yellow bats were 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 the southern yellow bat through 1995 (Kurta and Lehr 1995).

The western yellow bat is a medium- to large-sized bat, with pelage that is yellowish-buff to light brownish, with the fur tipped with gray or white. The forearms range from 41.5 to 49.0 millimeters (mm), the weights range from 9.2 to 22.5 grams on average, and the wingspan ranges from 33.5 to 35.5 centimeters. The ears are shorter than many other species, but the length is larger than the width (17.0 mm long). The anterior half of the dorsal surface of the uropatagium is well furred, while the posterior half is bare or almost bare (AGFD 2003). Western yellow bats differ from southern yellow bats 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 the short, round ears and long tail membrane, with at least the anterior portion well furred. *Lasionycteris* spp. hair is black with silver tips, while the yellow bat never has black hairs. The uropatagium is completely furred in other *Lasiurus* spp. found in Arizona. The hoary bat (*Lasiurus cinereus*) is larger (forearm 50.0–57.0 mm), has black edging around the ears, and has a mahogany brown pelage that is distinctly silver tipped. The western red bat is smaller (forearm 38.0–43.0 mm) and has a reddish pelage. Also, the yellow bat is known to be sexually dimorphic in size, with the female being slightly larger (forearm 2 mm larger) (AGFD 2003).

Western yellow bats are solitary and thought to not migrate (AGFD 2003). However, Williams (2001) found that populations in southern Nevada declined 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 winter. In Arizona, Cockrum (personal communication *in* AGFD 2003) found that males were generally found in spring and summer, and females were found from mid-winter 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 1995). 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 and Lehr 1995). It is unknown if western yellow bats breed in Arizona, but reproductive females were common in southern Nevada (AGFD 2003; 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. Western 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 western yellow bats in Moapa Valley, Nevada, were more active acoustically in riparian woodlands rather than other habitats in the area such as marsh, riparian shrubland, and mesquite bosque. 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). Their roosting height can range from 2.2 meters 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 the fur coloration of the species, although the bats will use any tree that gives them enough cover to be hidden while roosting. In Arizona, western yellow bats are found at elevations from 168 to 1830 meters (AGFD 2003). Along the LCR, yellow bats have been recorded at a cottonwood (*Populus* spp.) revegetation site at the Imperial National Wildlife Refuge and a dense palm grove just north of Parker, Arizona (Brown 2006). Results of a radio telemetry study indicate that yellow bats captured within cottonwood-willow riparian areas would almost always roost in nearby palm trees, usually in the yards of houses that were interspersed in areas of agricultural use. The results of this same study indicate that yellow bats selected palm trees that were at least 5.8 meters tall but usually above 9 meters (Mixan et al. 2014).

## Threats

Known predators of western yellow bats include domestic cats and dogs as well as barn owls (*Tyto alba*) (Kurta and Lehr 1995). Predators of other foliage-roosting bats include birds of prey, greater roadrunners (*Geococcyx californianus*), and opossums (*Didelphis virginiana*) (Shump, Jr. and Shump 1982). Woodpeckers (Picidae) and raccoons (*Procyon lotor*) have been observed disturbing other tree-roosting species at their roosting sites (Sparks et al. 2003). Human-caused threats include barbed wire fences and vehicles. The negative image some people have about bats has caused them to harass or even harm bats (Baker 1983 *in* Myers and Hatchett 2000; 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 2001).

Wind energy facilities have become a more recent threat to migrating bats of the *Lasiurus* genus. Bat fatalities occur at these facilities, as they may collide with wind turbine blades or experience barotrauma (a condition in which a rapid air pressure reduction near rotating blades of the turbine causes tissue damage in the lungs due to the rapid expansion of air in the lungs that does not accommodate exhalation) (Baerwald et al. 2008). Migrating species are the most common bats affected by wind energy facilities, with the vast majority of fatalities occurring in late summer or fall. Most of these species are tree-roosting bats, including both eastern and western red bats (Kunz et al. 2007). Some theorize that tree-roosting bats, which mate during this time period, are looking for the tallest trees in the landscape to find a mate, and wind energy facilities may be providing artificial stimuli that attract them (Cryan 2008; Cryan and Barclay 2009).

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# Yuma Hispid Cotton Rat (*Sigmodon Hispidus Eremicus*)

## INTRODUCTION

Yuma hispid cotton rats (*Sigmodon hispidus eremicus*) are a geographically isolated subspecies of hispid cotton rats (*S. hispidus*). They are 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. Yuma hispid cotton rats are one of two species of *Sigmodon* known to occur along the LCR; the other is Colorado River cotton rats (*Sigmodon arizonae plenus*), which are a subspecies of Arizona cotton rats (*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 deoxyribonucleic acid (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 Colorado River cotton rats instead of Yuma hispid cotton rats.

## LEGAL STATUS

Yuma hispid cotton rats are a Federal C2 candidate taxa. They are also in the International Union for Conservation of Nature Red List category and are considered a lower risk, near-threatened species. In California, they are a taxa of concern (Hafner and Kirkland 1998).

## DISTRIBUTION

The distribution of Yuma hispid cotton rats 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 Yuma hispid cotton rats may have historically occurred in the western part of the Gila River Valley east of Yuma, no evidence exists indicating that they are currently present in these areas (Hoffmeister 1986). Yuma hispid cotton rats have been trapped in Mexico close to the border, near Yuma, Arizona (Blood 1990). It is presumed that they were once much more prevalent in the Colorado River Delta area before changes on the river brought about the end of riverflows reaching the delta (Hafner and Kirkland 1998).

Hispid cotton rats are very widespread, and their range includes northern South America, Mexico, Central America, and the Southeastern and south-central

United States. However, the Yuma hispid cotton rat 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 Yuma hispid cotton rats is limited to areas near Yuma, Arizona, but may overlap with that of Colorado River cotton rats, which also occur along the LCR; the southern extent of their range is unknown. The northern extent of the range of the subspecies is presumed to be the Palo Verde Mountains, but there are not enough data to assume that no overlap occurs between the two species (Blood 1990). Results of a more recent study of the two species along the LCR showed no evidence of range overlap. Yuma hispid cotton rats were found as far north as the Imperial National Wildlife Refuge, and Colorado River cotton rats were not found south of the Cibola National Wildlife Refuge. The area in between was also surveyed but to a lesser extent due to difficult access and lack of suitable habitat in that area of the river (Neiswenter 2010). 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). More recent surveys have confirmed Colorado River cotton rats along the LCR near the town of Gadsden, Arizona, as well as on the western Gila River near where it crosses the AZ-95 highway (unpublished data).

Hoffmeister (1986) saw both species (hispid cotton rats and Arizona cotton rats) together in laboratory situations and observed hispid cotton rats to be subordinate. In the laboratory, the two species were aggressive, with Arizona cotton rats taking a dominant position over the hispid cotton rat by gaining first access to food and other resources. Hoffmeister hypothesizes 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 Yuma hispid cotton rats. 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).

Hispid cotton rat have small home ranges, with females typically occupying home ranges of 0.1 to 0.3 hectare and males occupying home ranges of 0.4 to 0.5 hectare (Hawthorne 1994). Radio telemetry study results have shown that hispid cotton rats use these home ranges in a systematic manner, over multiple days, which may allow the species to use 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 use runways through dense herbaceous growth, and nests are built of woven grass. The population density is regulated by avian predators; mammal predation is considered to be incidental. Some of the principal competitors for resources of 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 their distribution possibly overlapping that of the Colorado River cotton rat range and the inability to distinguish Yuma hispid cotton rats from Colorado River cotton rats 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 Yuma hispid cotton rats from Colorado River cotton rats (Blood 1990; Sevringhaus and Hoffmeister 1978; Zimmerman 1970). An Arizona cotton rat was bred in a laboratory with a hispid cotton rat and only infertile young were produced (Zimmerman 1970), which would suggest that hybridization is rare, if it occurs at all.

Peppers and Bradley (2000) conducted genetic analyses on eight subspecies of *Sigmodon* and determined that Yuma hispid cotton rats are genetically similar to other subspecies of *Sigmodon,* which occur in New Mexico and Oklahoma. Yuma hispid cotton rats were 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 hispid cotton rat subspecies. All hispid cotton rats captured during the Neiswenter (2010) study were of a single haplotype that appeared to be unique to the *eremicus* 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. Average litter sizes are 5.6 young, and 1 captive female in a laboratory situation was recorded to have produced

9 litters in a 10-month period (Hoffmeister 1986). Breeding starts after 2–3 months of age, and the average lifespan is 6 months (Cameron and Spencer 1981).

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

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# EVALUATION SPECIES

# California Leaf-nosed Bat (*Macrotus californicus*)

## LEGAL STATUS

California leaf-nosed bats (*Macrotus californicus)* are not federally listed as threatened or endangered. The Bureau of Land Management (BLM) lists them as a sensitive species in California (BLM 2004). California recognizes them as a mammalian species of special concern (Williams 1986). This species was included in a draft list of Arizona Wildlife of Special Concern by the Arizona Game and Fish Department (AGFD) (AGFD 2001). California leaf-nosed bats are a Nevada Species of Conservation Priority and protected and considered sensitive (Nevada Department of Wildlife 2005). The Western Bat Working Group (1998) lists them as a species of "Red or High" priority, the highest priority available. This species is included on the International Union for Conservation of Nature Red List of Threatened Species as vulnerable – its third highest rating (Chiroptera Specialists Group 1996).

## DISTRIBUTION

### Historical Range

The historical range of California leaf-nosed bats included records from San Diego and Riverside Counties, California, eastward to Tombstone, Arizona, and south into Baja California and Sonora, Mexico, with the center of their distribution appearing to be the location of their 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 mile west of Lake Mojave. At least three mines that were known roost sites were inundated by water with the formation of Lakes Mead and Mojave (O'Farrell 1970). Later, this 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; Constantine 1961).

### Current Range

The current range of this species 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 www.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 it was recorded at two different sites during a 2002–03 bat survey in southwest San Diego County (Stokes et al. 2003). All records in Arizona were from below 4000 feet (1220 meters) in elevation, with most below 2500 feet (7625 meters) (AGFD 2001).

## Populations Within the LCR MSCP Planning Area

The Lower Colorado River Multi-Species Conservation Program (LCR MSCP) planning area 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 (Lakes Mead, Mojave, and Havasu) along the lower Colorado River (LCR). The lower ends of the Virgin and Bill Williams Rivers, which are LCR tributaries, are included in the LCR MSCP planning area (Bureau of Reclamation 2004).

California leaf-nosed bats have known populations all along the LCR. In Nevada, they have 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; O'Farrell Biological Consulting 2006). In the Lake Mead National Recreation Area, populations have been found roosting in three mines (Brown 2006). A California leaf-nosed bat was captured on the Arizona side of the recreation area at a mine near Katherine Landing, which is now closed (Cockrum et al. 1996). The Havasu National Wildlife Refuge (Havasu NWR) is the location of at least two mines that support leaf-nosed bats (Brown 2006; Cockrum et al. 1996). The Havasu NWR is located along approximately 30 miles of the Colorado River from Needles, California, to Lake Havasu City, Arizona. The Island Unit bridge on the Cibola National Wildlife Refuge (Cibola NWR) was found to be an important night roosting spot for this 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). The Cibola NWR is 15 miles south of Blythe, California, along the Colorado River. At least seven mines along the Bill Williams River contain colonies of these bats, ranging from 100 to 1,000. (Brown 1996). The Bill Williams River empties into Lake Havasu north of Parker, Arizona. During a survey done on the Arizona side of the Imperial National Wildlife Refuge (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, the California myotis (*Myotis 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 the Cibola NWR and the Imperial NWR, by the AGFD in 1995. Leaf-nosed bats were present at 8 sites, and there was evidence of bat use at 20 sites, 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 along the LCR (Brown 2006). California leaf-nosed bats have been captured at multiple habitat restoration sites, including the Beal Lake Conservation Area within the Havasu NWR; the ‘Ahakhav Tribal Preserve near Parker, Arizona; Palo Verde Ecological Reserve near Blythe, California; Cibola Valley Conservation Area; Cibola NWR Unit #1 near Cibola, Arizona; Imperial NWR Nursery; Pratt Restoration Site near Laguna Dam; and Yuma East Wetlands near Yuma, Arizona (Calvert 2010, 2012; Diamond 2012; unpublished data).

# LIFE HISTORY

## General Description

California leaf-nosed bats were originally classified as their own species (Grinnell 1918). Later, they were classified as a subspecies of Waterhouse’s leaf-nosed bats (*Macrotus waterhousii californicus*) of a leaf-nosed bat found in Mexico, Guatemala, and the Caribbean (Anderson 1969). They regained species status when Davis and Baker (1974) found that Waterhouse’s leaf-nosed bats had a chromosomal diploid number of 2N = 46, while California’s leaf-nosed bats 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. The results of a renal (kidney) morphology study show that leaf-nosed bats can use drier habitats than Waterhouse’s leaf-nosed bats 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 the gray-furred, medium-sized California leaf-nosed bat. The ears are large (29–38 millimeters [mm]) and joined near the base. The tail extends past the uropatagium 5–10 mm. The forearm measures 46–55 mm, and the weight varies from 12 to 22 grams. Mexican long-tongued bats (*Choeronycteris mexicana*) and lesser long-nosed bats (*Leptonycteris curasoae*) 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 do (Hoffmeister 1986; Kays and Wilson 2002; AGFD 2001). Their total lifespan is not known; however, one California leaf-nosed bat 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 caves or mines that maintain high temperatures (greater than 28 degrees Celsius); many of these caves are geothermally heated.

## Breeding

Females gather into maternity colonies in 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 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 embryos is delayed until the following spring. A single young is born between mid-May and early July (Hoffmeister 1986; Berry and Brown 1995; AGFD 2001).

## Diet

Echolocation and visual detection are used by leaf-nosed bats to locate prey; the latter is used more in the winter months to save energy (Bell et al. 1986). They 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; Hoffmeister 1986). Brown and Berry (2004) discovered a California leaf-nosed bat at the 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 by leaf-nosed bats usually takes place in dry desert washes, 3–6 miles from their roost. In winter, this distance decreases to 1/2 mile from their roost (Brown et al. 1993; Brown 2005). Desert wash plant communities include ironwood (*Olneya tesota*), Palo verde (*Cercidium* spp.), mesquite (*Prosopis* spp.), catclaw (*Acacia greggii*), and smoketree (*Psorothamnus spinosa*) (Castner et al. 1995a). Huey (1925) found evidence of leaf-nosed bats using riparian areas as well. Along the Muddy River, in Moapa Valley, Nevada, a California leaf-nosed bat was equally detected acoustically in four distinct habitat types: riparian marsh, mesquite bosques, riparian woodlands, and riparian shrublands. The woodlands consisted of Fremont cottonwood (*Populus fremontii*), velvet ash (*Fraximus velutina*), Goodding's willow (*Salix gooddingii*), and Washington fan palms (*Washingtonia filifera*). The 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 California leaf-nosed bats forage during the entire year, foraging habitat in close proximity to roosting sites may be more important during the winter months (Brown et al. 1993; Brown 2006). While night roosts consist of shallow caves in natural situations and manmade structures, including mining prospects, bridges and buildings are also readily used near foraging habitat (Huey 1925; Hatfield 1937; 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 a California leaf-nosed bat is from evidence of a dentary bone in barn owl (*Tyto alba*) pellets in Sonora, Mexico (Bradshaw and Hayward 1960). A leaf-nosed bat was found impaled on a barbed wire fence outside of a mine entrance near the Bill Williams River, likely by a loggerhead shrike (*Lanus ludovicianus*), which was found flying away from the mine entrance as people approached (unpublished data). Bats, in general, are preyed upon by a number of different animals, although most of them 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 (*Procyon lotor*), weasels (*Mustela* spp.), predatory song birds (Passeriformes), frogs (Anura), large spiders (Araneae), and even other bats (Fenton 2001). While humans are not predators of leaf-nosed bats, the negative image some people have about bats has caused them to harass or even harm them. (Fenton 1997).

Disturbance and closure of roost mines are the greatest threats to California leaf-nosed bats (Brown 2005). Disturbance may cause abandonment of roosts (AGFD 2001). The best way to keep a mine open for leaf-nosed 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

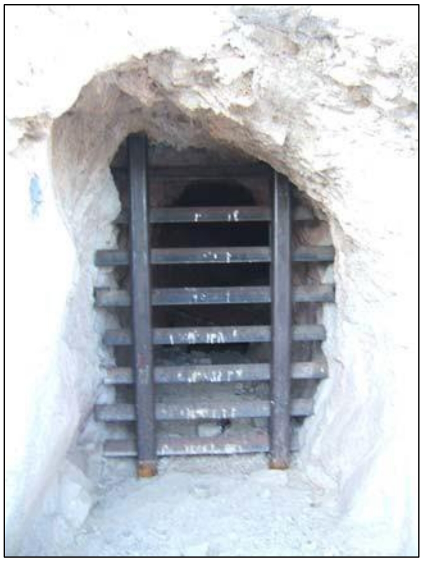

**Figure 1.—Example of a gated mine at the Salt Creek Hills Mine near Baker, California.**

(figure 1). Because the bats are restricted by 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).

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# Colorado River Toad (*Bufo = Incilius alvarius*)

## LEGAL STATUS

Colorado River toads (also known as Sonoran Desert toads) (*Bufo alvarius*) are not federally listed. They are listed as threatened in New Mexico (Degenhardt et al. 1996). NatureServe ranks this species as secure on a national and global level and as secure in Arizona, imperiled in New Mexico, and possibly extirpated in California (NatureServe 2006).

## DISTRIBUTION

### Historical Range

The westernmost record of Colorado River toads 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 1951); southwestern New Mexico (Cole 1962); northwest of the junction of Arizona Highway 71 on U.S. 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 Colorado River toads in California extended along the flood plain of the lower Colorado River (LCR) and in the southern Imperial Valley. Historically, they were documented along the LCR from Fort Yuma to the Blythe-Ehrenberg Region (Fouquette 1968; Vitt and Ohmart 1978). Their range likely extends 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 Colorado River toads.

## Current Range

Colorado River toads are currently restricted to the Sonoran Desert in the 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 7 miles west of Guamuchil, Sinoloa Mexico (Fouquette 1968, 1970; Riemer 1955). This species is found at elevations ranging from above sea level to 1600 meters (Cole 1962).

The total adult population size for this species is unknown (NatureServe 2006). Colorado River toads are common throughout their range in Arizona but populations have 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 July 31, 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 kilometers south of Blythe, California (Anderson and Ohmart 1982 *in* Jennings and Hayes 1994; J. Rorabaugh, personal communication *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, personal communication *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 Lower Colorado River Basin in the vicinity of Winterhaven, Palo Verde, Bard, Ferguson Lake, Goose Flats, and the Cibola National Wildlife Refuge in which Colorado River toads were not detected (King and Robbins 1991). Extensive surveys downstream from Imperial Dam during the 1980s and 1990s were conducted, and this species was not detected (J. Rorabaugh, personal communication *in* SAIC/Jones & Stokes 2003). A survey was conducted in August 1999 at three sites within the Cibola National Wildlife Refuge (Mitchells Camp, Walters Camp, and the Anderson and Ohmart Dredge Spoil Revegetation Site) and around Parker Dam (up to Black Meadow Landing). Colorado River toads were not detected during these surveys (J. Rorabaugh, personal communication *in* SAIC/Jones & Stokes 2003). Recent surveys along the LCR from Needles, California, south to Yuma, Arizona, also did not find this species on the main stem; however, they were detected on the Bill Williams River within the vicinity of Planet Ranch (Cotten and Leavitt 2014).

Three hybrids between Colorado River toads and woodhouse’s toads (*B. 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 was unknown (Gergus et al. 1999). Fossil evidence suggests some genomic compatibility has

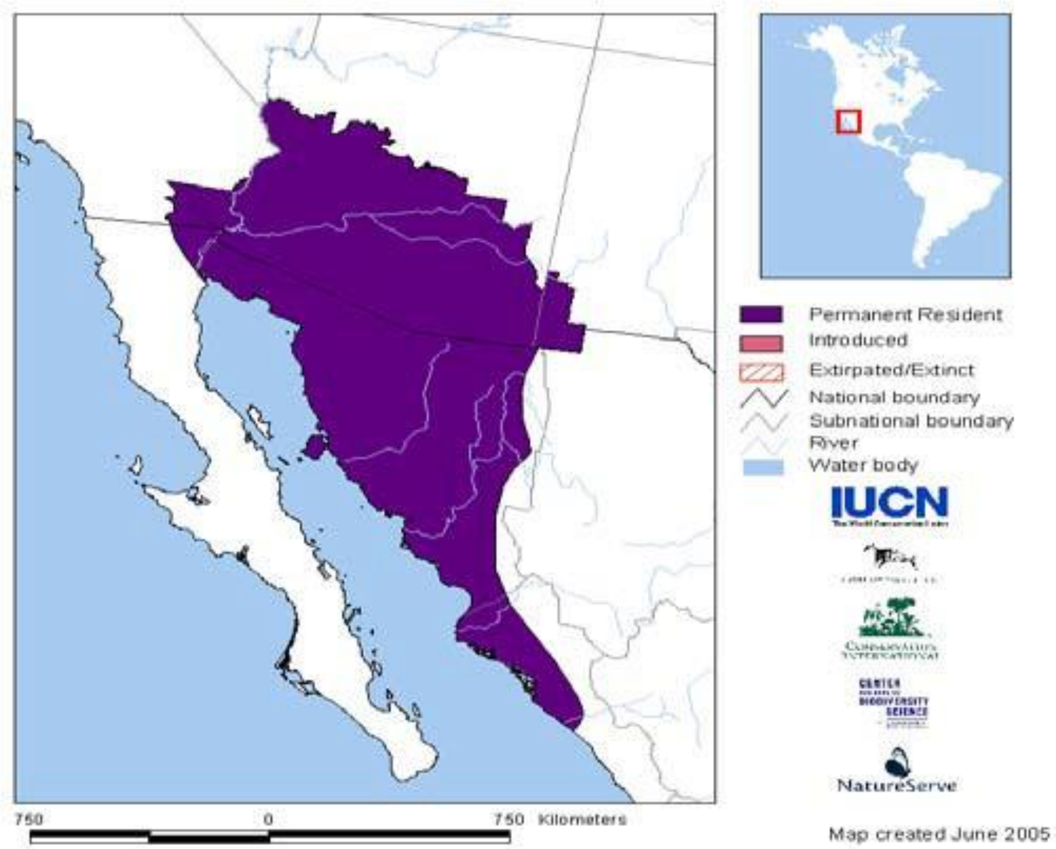

Figure 1.—Range of Colorado River toads.

been retained between the Colorado River and woodhouse's toads despite at least 6 million years of independent evolution (Gergus et al. 1999). Hybridization between the two species may be due to the increase of females actively searching for males during the mating period (Gergus et al. 1999).

# LIFE HISTORY

## General Description

The holotype of Colorado River toads 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). They are nocturnal toads in the family Bufonidae. Common names for this species are the Colorado River toad and the Sonoran desert toad (Brennan and Holycross 2006). Colorado River toads are large anurans with a snout-to-vent length (SVL) ranging from 110 to 187 millimeters (mm). This species 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 the nostril to the tympanum; the width is less than half the length. Two distinct traits of this species are one to four conspicuous, whitish, rounded tubercles 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. SVL and age, estimated by lines of arrested growth, were not significantly correlated in a population in Maricopa County, Arizona (Sullivan and Fernandez 1999). Growth rates appear to decrease with age (Sullivan and Fernandez 1999).

Tadpoles of this species have a brassy coloration, rounded tail, and flattened body, and they can reach a size of 57 mm total length (Deganhardt et al. 1996).

## Breeding

Colorado River toads breed from May through August in ponds, slow-moving streams, temporary pools, or manmade structures that hold water (Stebbins 1985; NatureServe 2006). In Arizona, they usually breed 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 this species to initiate breeding and chorusing activity (Sullivan and Fernandez 1999; Sullivan and Malmos 1994). Breeding and chorusing activity usually occurs 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 Colorado River toads over a 6-year time period in the absence of successful breeding through many seasons.

The clutch size of Colorado River toads is between 7,500 and 8,000 eggs per female. Eggs are 1.6 mm in diameter, 5–7 centimeters 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, Jr. et al. 2005). Eggs are deposited in shallow pools of water (Fouquette, Jr. 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 metamorphose into a froglet (Musgrave and Cochran 1930; Brennan and Holycross 2006). When compared to other frogs and toads, Colorado River toads develop from zygotes to hatchlings at a remarkably fast rate (Musgrave and Cochran 1930; Brennan and Holycross 2006). The eggs of this species have different characteristics than other members of the *B. boreas* group in that they completely lack 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, Jr. et al. 2005). Males use two strategies for pairing with females, including active searching and stationary calling from shallow water. The strategy chosen appears to be influenced by the size of the 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 Colorado River toads does not play a role in breeding behavior; however, Sullivan and Malmos (1994) demonstrate in preliminary trials that females were attracted to the call of males. Further research on mate selection techniques needs to be conducted.

The Colorado River toad’s advertisement call consists of an emphasized note followed by three progressively weaker notes. The fundamental frequency of the advertisement call is 1,096 cycles per second, and the duration is approximately 0.7 second in length (Blair and Pettus 1954). Sound pressure levels of advertisement calls are approximately 88 decibels at about 0.5 meter, 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). The frequency or duration of advertisement or release calls is not related to body size or body temperature of individuals (Sullivan and Malmos 1994). The pulse rate of advertisement calls increases as the body temperatures of individuals increase (Sullivan and Malmos 1994). The pulse rate of release calls decreases as the body temperatures of individuals increase (Sullivan and Malmos 1994). The Colorado River toad’s arytenoid cartilages are believe to be activated only during production of release calls (Sullivan and Malmos 1994).

## Diet

Colorado River toad adults are active foragers and feed on invertebrates, lizards, small mammals, and amphibians (Brennan and Holycross 2006). A stomach content analysis of five specimens and an intestinal analysis of one specimen produced members of the following orders, from most abundant to least abundant: beetles (Coleoptera); wasps, ants, and bees (Hymenoptera); termites (Isoptera); sun spiders (Solifugae); true bugs (Hemiptera); butterflies and moths (Lepidoptera); spiders, mites, and scorpions (Arachnida); grasshoppers, locusts, and crickets (Orthoptera), millipedes (Spirobolida); and centipedes

(Scolopendromorpha) (Cole 1962). Tadpoles are believed to be algivorous and omnivorous. Colorado River toads are able to eat prey that is protected by sting mechanisms or defensive secretions (Cole 1962).

## Habitat

Colorado River toads are semiaquatic toads that occur primarily in desert habitat, including mesquite-creosote lowlands, but also inhabit 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 meters (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 (*Pinus cembroides*), alligator juniper (*Juniperus deppeana*), various oaks (*Quercus* spp.), and a native grassland in Chihuahuan desert scrub. Cole (1962) observed this species associated with agave (*Agave* spp.), joint fir (*Ephedra* spp.), mesquite (*Prosopis* spp.), Russian thistle (*Salsola* spp.), yucca (*Yucca* spp.), snakeweed (*Gutierrezia* spp.), and grasses. This species may have also expanded its range to agricultural areas when large-scale conversion of native habitat to agriculture took place. The study by Cotten and Leavitt (2014) found that the toads were primarily using sandy creosote flats that were adjacent to the Bill Williams River.

The breeding habitat of Colorado River toads includes seasonal and permanent pools (Fouquette 1970; NatureServe 2006; Wright and Wright 1949 *in* Fouquette, Jr., et al. 2005). MacMahon (1932 *in* King and Robbins 1991) describe preferred Colorado River toad habitat as damp areas near permanent springs or manmade watering holes. They are also known to use 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. This species 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) describe agricultural drains, dam seepages, irrigation canals, and backwaters along the LCR as “marginal habitat” for this species.

The habitat for Colorado River toads when dormant, or their refugium during active periods, includes subterranean shelters such as rodent burrows, rock outcrops, or hollows under watering troughs (Wright and Wright 1949 *in* Fouquette, Jr., et al. 2005; Lowe 1964 *in* Fouquette, Jr., et al. 2005; D. Beck, unpublished data *in* Fouquette, Jr. et al. 2005).

The general habitat preferences for Colorado River toads 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 under 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 Conservation Area 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 (*Pseudacris regilla*) were observed breeding in these areas since April 2006. Other habitat creation projects may provide similar habitat to the Pacific tree frog and other species of frogs and toads.

## Defense Behavior

Colorado River toads are capable of emitting a poisonous substance (toxic indolealklamines) from their parotoid glands (Hanson and Vial 1956; Musgrave and Cochran 1930; Erspamer et al. 1967 *in* Fouquette, Jr. et al. 2005, Cei et al. 1968 *in* Fouquette, Jr. et al. 2005). 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 (*Mephitis mephitis*) in a laboratory setting and showed no effect on a domestic cat or a domestic dog. 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 (Hanson and Vial 1956). Musgrave and Cochran (1930) reported a fox terrier dying after biting into a Colorado River toad. Musgrave and Cochran (1930) also reported that facial contact with one of these toads caused a police dog to be paralyzed for approximately 60 minutes. An effective defense posture of this species is to face the predator with its head, dorsum, and parotoid glands and make a hissing sound (Hanson and Vial 1956).

## Biology

Colorado River toads are sympatric with spadefoot toads (*Scaphiopus* spp.), great plains toads (*B. cognatus*), red-spotted toads (*B. punctatus*), and woodhouse toads (Blair and Pettus 1954; Sullivan and Malmos 1994; Wright and Wright 1949 *in* Fouquette, Jr. et al. 2005). The age of individuals of Colorado River toads 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, Jr. et al. 2005).

Colorado River toads migrate short distances between breeding and non-breeding habitats, but no studies have been conducted on the length of these migrations (NatureServe 2006; Fouquette, Jr. et al. 2005). Individuals may migrate several hundred meters from permanent to seasonal pools following heavy rains (Wright and Wright 1949 *in* Fouquette, Jr. et al. 2005). Little research has been conducted on the home range of this species (Fouquette, Jr. 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. Colorado River toads are 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). The accuracy of calling surveys and frogloggers is dependent on the ability of observers to identify calls made by amphibian species. Egg mass surveys are used with pond and pool breeders and 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 using the habitat, and the proportion of habitat occupied by a 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 through 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). An accurate and precise determination of leopard frog population sizes requires the use of mark-recapture methods (Donnelly and Guyer 1994).

## Threats

Habitat loss and alteration in the LCR region likely have had an impact on Colorado River toad 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). Non-native species that have an effect on many native species, such as bullfrogs (*Rana catesbieana*) and spiny softshell turtles (*Apalone spinifera*), may also affect Colorado River toads along the LCR. Illegal collection of Colorado River toads 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 the habitat requirements of Colorado River toads, the specific reasons why they have declined along the LCR are unknown (Jennings and Hayes 1994). Furthermore, the historical abundance of this species in this region is not quantitatively known, so the extent of decline in this region cannot be verified. Cole (1962 *in* Fouquette, Jr. et al. 2005) reports Colorado River toads 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 this species are discovered along the LCR or a translocation program is initiated, habitat requirements and factors negatively affecting their populations must be determined.

Predators of this species include raccoons (*Procyon lotor)*, possibly birds, other mammals, and reptiles (Wright 1966). Wright (1966) observed a raccoon ripping open the abdominal cavities of five Colorado River toad adults and consuming the contents of the cavities but leaving the dorsal portion of the carcasses. Hanson and Vial (1956) observed that the defensive posture and skin toxins protect it from striped skunks. It is likely that adult Colorado River toads are safe from most predators due to the toxicity of their parotoid secretions and their large size; however, no research study has been conducted on predators of this species.

Parasites known to affect Colorado River toads include *Aplectana itzocanensis, Oswaldocruzia pipiens, Physaloptera* spp., *Physocephalus* spp., and *Rhabdias americanus* of the phyla Nemotoda and *Nematotaenia dispar* of the family Cestoidea (Goldberg and Bursey 1991).

Although populations of Colorado River toads 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 (T. Jones 2005, personal communication).

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# Desert Pocket Mouse (*Chaetodipus penicillatus sobrinus*)

## INTRODUCTION

Desert pocket mice (*Chaetodipus penicillatus*) occur throughout the deserts of the Southwestern United States and northwestern Mexico. Two subspecies occur along the lower Colorado River (LCR): *Chaetodipus penicillatus sobrinus* and *C. p. penicllatus*. *C. p. sobrinus* is the subspecies that is covered as an evaluation species under the Lower Colorado River Multi-Species Conservation Program (LCR MSCP). Desert pocket mice were previously classified under the scientific name *Perognathus penicillatus* and were referred to by this scientific name prior to 1983.

## LEGAL STATUS

*C. p. sobrinus* is an evaluation high priority subspecies under the Clark County Multi-Species Habitat Conservation Plan. They are currently an evaluation species under the LCR MSCP Habitat Conservation Plan.

## DISTRIBUTION

Desert pocket mice occur in creosote bush and xeric riparian communities of the southwestern deserts of North America. Their northern range encompasses areas from southeastern California, southern Nevada, and extreme southwest Utah. To the south, this species occurs to southeastern Baja California, Mexico, and the northern two-thirds of Sonora, Mexico (Mantooth and Best 2005).

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 (Z. Marshall 2005, personal communication). The other subspecies that occurs along the LCR, *C. p. penicillatus*, has a wider range and occurs from Topock Marsh along 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).

*C. penicillatus* have been caught in the Beal Lake Conservation Area within the Havasu National Wildlife Refuge and are frequently captured in the Big Bend Conservation Area near Laughlin, although the subspecies is not known (Hill 2012). If species presence is to be verified for future trapping, toe/ear clips can be taken and sent to a genetics facility at the University of Nevada, Las Vegas.

# LIFE HISTORY

## General Description

*C. p. sobrinus* is the largest of the two subspecies occurring along the LCR but is not strongly differentiated from *C. p. penincillatus* (Hoffmeister 1986). *C. 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).

*C. p. sobrinus* is the northernmost subspecies of pocket mice. 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 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). The maximum lifespan of individuals was 16 months, and the annual turnover of the population was estimated to be between 87 and 90 percent (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). *C. 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 the late spring and early summer (Mantooth and Best 2005). In the northern part of the range, activity decreases greatly in the winter months, with relatively few individuals captured from November to April (Bateman 1967; Van De Graaff 1975; Andersen and Nelson 1999). This species may be active year round in the southern part of their range (Jameson and Peeters 2004). In areas where winter activity decreases, some individuals enter into short periods of torpor during energetic crises (French 1993).

## Breeding

Their breeding season occurs from April to August, with peak reproductive activity occurring in June, when 95 percent of males are capable of reproduction (Hoffmeister 1986; Mantooth and Best 2005). *C. penicillatus* build sphere-shaped nests of dry grass, dug to a depth of roughly 18 centimeters (Hoffmeister 1986). Their gestation period is 26 days or more (Eisenberg and Isaac 1963; Wilken and Ostwald 1968). A litter size averages 3.38 (Van de Graaff 1975).

## Habitat

*C. penicillatus* occur in desert areas with coarse vermiculite soils and clumped brush habitat (Price 1984; Mantooth and Best 2005). 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 2005). They are the only species of heteromyid rodent commonly found in riparian woodland or salt cedar (*Tamarix* spp.) habitats (Stamp and Ohmart 1979). *C. penicillatus* is fully independent of exogenous water (Grubbs 1974). While they prefer areas with shrubby canopy cover, they forage into open areas up to 4 meters from cover (Rosenzweig 1973).

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# Lowland Leopard Frog
# (*Rana = Lithobates yavapaiensis*)

## LEGAL STATUS

The U.S. Forest Service and the Bureau of Land Management have listed lowland leopard frogs (also known as San Felipe and Yavapai leopard frogs) (*Rana yavapaiensis*) as a sensitive species (New Mexico Game and Fish Department 2004). They are listed as an endangered species and are provided full protection in New Mexico (New Mexico Game and Fish Department 2004). They are listed as a species of concern in Arizona (Arizona Game and Fish Department [AGFD] 2001). This species is extirpated from California (Vitt and Ohmart 1978) and from all but Hildago County in New Mexico (Scott, Jr. 1992; Jennings 1995). Lowland leopard frogs are listed in the Special Protection Category by the Mexican Government (NatureServe 2006). NatureServe ranks the status of this species as apparently secure on a national and global level and as apparently secure in Arizona, presumed extinct from California, and critically imperiled in New Mexico (NatureServe 2006).

## DISTRIBUTION

### Historical Range

Lowland leopard frogs were historically distributed along the lower Colorado River (LCR) and its 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, downstream to near the mouth of the Colorado River in Mexico (Jennings and Hayes 1994a, 1994b; Vitt and Ohmart 1978). In California, their historical range extended discontinuously from San Felipe Creek, near its junction with Carrizo Creek, and eastward through the Imperial Valley to the entire LCR (Jennings and Hayes 1994a; California Department of Fish and Game 1994). 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, this species was found at 28 locations in California, 14 locations in New Mexico, and 302 locations in Arizona (NatureServe 2006).

Historical records for lowland leopard frogs in Arizona have been noted in Mohave, Yavapai, Coconino, La Paz, Maricopa, Gila, Pinal, Graham, Greenlee, Yuma, Santa Cruz, and Cochise Counties. In California, this species has been recorded in San Bernardino, Riverside, and Imperial Counties.

In New Mexico, this species has been recorded in Catron, Grant, and Hildago Counties (Vitt and Ohmart 1978; Clarkson and Rorabaugh 1989; Jennings and Hayes 1994b; Sredl et al. 1997b).

Lowland leopard frogs were historically present along the LCR and in its natural overflow lakes and tributary streams (Jennings and Fuller 2004; Vitt and Ohmart 1978; Clarkson and Rorabaugh 1989). Stebbins (1951) found this species present below Imperial Diversion Dam. Grinnell and Camp (1917) report individuals in Riverside County, California, along the Colorado River, north at least to Riverside Mountain. Slevin (1928) reports individuals in Yuma County, Arizona. Observations indicate that the species expanded their 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

Lowland leopard frogs occur in the southern half of Arizona as well as adjacent parts of Sonora, Mexico (Platz and Frost 1984). This species remains well represented in interior Arizona, south and west of the Mogollon Rim (Clarkson and Rorabaugh 1989; Sartorius and Rosen 2000). They are present in every county in Arizona except Apache and Navajo, with 57% of all localities occurring in Gila, Maricopa, and Yavapai Counties (Sredl et al. 1997b). Eighty percent of extant lowland leopard frog habitat is located in the Gila River drainage (Salt, Upper Gila, Verde, and 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). This species was detected in Arizona in 1997 at 43 of 115 historical sites surveyed. Populations were detected at 61 sites where historical records for lowland leopard frogs did not exist (Sredl et al. 1997b). This species occurs in several canyons in Saguaro National Park (Parker 2005). Populations of leopard frogs confirmed to be lowland leopard frogs were discovered along the Colorado River in Surprise Canyon (west of Separation Canyon) in the western Grand Canyon in the spring of 2004 (Gelciz and Drost 2004). This population was found 4 miles (6.4 kilometers [km]) up the canyon in a small pool (1 by 2 meters) that had a sparse growth of cattails (*Typha*) around the edge (Gelciz and Drost 2004). This was the first recent observation of this species in this location; however, this species was known to historically occur in this section of the river. This observation extends the current distribution of lowland leopard frogs further north (Gelciz and Drost 2004). Populations are declining in southeastern Arizona (AGFD 2001). Surveys by Vitt and Ohmart (1978), Clarkson and Rorabaugh (1989), and Jennings and Hayes (1994b) found extant populations of this species in only two localities in southwestern Arizona.

No lowland leopard frogs have been found in California; therefore, they are believed to be extirpated from this State. The most recent record for this species in California was collected in 1965 from an irrigation ditch east of Calexico (Jennings and Hayes 1994b). Lowland leopard frogs in New Mexico are 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 lowland leopard frogs, were confirmed to be relict leopard frogs (*R.onca*) (Jaeger et al. 2001).

Lowland leopard frogs are sympatric at intermediate elevations (1180–1700 meters) with Chiricahua leopard frogs (*R. 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). Chiricahua leopard frog and lowland leopard frog hybrids are very rare (Platz and Frost 1984). Laboratory experimental crosses between Chiricahua leopard frogs and lowland leopard frogs show interspecific genetic compatibility. Mating call characteristics appear to be the pre-mating reproductive isolative that accounts for the limited occurrence of hybrid individuals between 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).

Lowland leopard frogs had become very rare along the LCR by the early 1960s and were considered extirpated by 1974 (Vitt and Ohmart 1978; Clarkson and Rorabaugh 1989; AGFD 2001). This species was not found in Imperial Valley, California; the LCR, Arizona-California; or the lower Gila River, Arizona, from 1983 to 1987 (Clarkson and Rorabaugh 1989). They are believed to be currently extirpated from the lower Gila and Colorado Rivers in Arizona and adjacent California (Sredl et al. 1997b). Lowland leopard frogs have been recently reported from approximately 7 miles (11.2 km) upstream of the confluence of the Colorado and Bill Williams Rivers, within the Bill Williams River National Wildlife Refuge (Jennings and Hayes 1994b; Clarkson and Rorabaugh 1989; AGFD 1998 *in* SAIC/Jones & Stokes 2003). Since then, two individual lowland leopard frogs have been found within the Bill Williams River National Wildlife Refuge, and a robust population has been discovered along the Bill Williams River just east of Planet Ranch (Cotten and Leavitt 2014).

## Current Abundance

Mark-recapture studies conducted at seven Arizona sites from 1991 to 1996 resulted in highly variable population estimates among sites and within sites, ranging from 19 to 1,806 individuals per site (Sredl et al. 1997a):

The Big Spring site, in Graham County, Arizona, was the only one of the seven sites where monitoring was conducted for all 6 years. The 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.

- The population size at the Tule Creek site, in Yavapai County, was estimated at 704 individuals in 1991, 887 individuals in 1992, and 1,806 individuals in 1993.
- The population size at Barnhardt Mesa, in Gila County, was estimated at 863 individuals in 1994.
- The population size at Alamo Canyon, in Pima County, was estimated at 41 individuals in 1991 and 41 individuals in 1992.
- The population size at Horsefall Canyon, in Cochise County, was estimated at 59 individuals in 1994.
- The population size at Reed Spring, in Gila County, was estimated at 19 individuals in 1992.
- The population size at Thicket Spring, in Bloody Basin, Yavapai County, was estimated at 73 individuals in 1991 (Sredl et al. 1997a).

At 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). At 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 winter (Sredl et al. 1997a).

# LIFE HISTORY

## General Description

J.E. Platz collected the first specimen of lowland leopard frogs in 1971 from Tule Creek in Yavapai, Arizona. The specimen was a single adult male described by Platz and Frost (1984) (California Department of Fish and Game 1994). Common names include lowland leopard frog, San Felipe leopard frog, and the Yavapai leopard frog. Lowland leopard frogs are a species in the *R. 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 the legs, lack of both vestigal oviducts and prominent external vocal sacs in males, and a dark reticulate pattern on the posterior surface of the 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 and 63.5 millimeters (mm), respectively (Platz and Frost 1984; Platz 1988). Lowland leopard frogs are morphologically most similar to Chiricahua leopard frogs and northwest Mexico leopard frogs (*R. magnaocularis*) (Platz and Frost 1984). Based on chromosomal and mitochondrial elements, northwest Mexico leopard frogs are the closest relative to lowland leopard frogs (Platz and Frost 1984; Pfeiler and Markow 2008). Lowland leopard frogs and northwest Mexico leopard frogs are quantitatively similar genetically but are qualitatively dissimilar in developmental compatibility with other species (Platz and Frost 1984). Sceletochronology of lowland leopard frogs indicates that individuals can live up to 3 years. Survivorship of adults and juveniles appears to be high in spring and summer and lower in fall and winter (AGFD 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

Lowland leopard frog 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. Lowland leopard frog populations are primarily connected by movement through drainages and not along straight lines (Goldberg et al. 2004). Goldberg et al. (2004) studied populations in Saguaro National Park and found distances between populations to range from 0.4 to 18.4 miles (0.7 to 29.7 km, respectively). Populations of this species 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 lowland leopard frog samples in the Tucson Basin can be attributed to variation among populations (Goldberg et al. 2004). Goldberg et al. (2004) conclude that populations of this species 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 are small and isolated (Sartorius and Rosen 2000).

Benedict et al. (2002) studied lowland leopard frog 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 suggest 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 2002). Source populations were found in the high elevation reaches, upstream of the main channels, and were the most genetically distinct and unique (Benedict 2002). More recent genetic analyses found that there are at least 19 haplotypes spread across most of the lowland leopard frog's range, but an additional 2 haplotypes were only found at 1 locality within Surprise Canyon within Grand Canyon National Park, indicating this population has been genetically isolated for a while (Oláh-Hemmings et al. 2009).

## Breeding

Lowland leopard frog 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 centimeters deep) 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 fall (September – October) often results in an overwintering population of larvae (Collins and Lewis 1979). The growth of tadpoles occurs in warm springs but is arrested in cold springs in other species of leopard frogs; this may occur with lowland leopard frogs (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 was high, but there was mortality among eggs of individual masses. The mortality usually occurred in the eggs that were 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 drier years than in wet years. Larvae metamorphose in 3 to 9 months (AGFD 2001). Lowland leopard frogs have an average SVL of 25–29 mm at metamorphosis (Platz 1988). Males reach sexual maturity when the SVL measures 53.5 mm. The size of females when they reach sexual maturity is unknown (AGFD 2001). Sex ratios are generally 1:1 (Sredl et al. 1997a).

Ruibal (1962) found that the temperature range of water for normal development of *R. pipiens* (lowland form) is between 11 and 29 degrees Celsius (°C). *R. pipien* egg masses (lowland forms) have not been found to be exposed to water temperatures greater than 25 °C (Ruibal 1962). Lowland leopard frogs produce a mating call that comprises many notes (typically 6–16), with the first note 0.5–2 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 the dominant frequency averages 1.8 kilohertz (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 this species is more similar to the mating call of northwest Mexico leopard frogs than to any other species of leopard frog (Platz and Frost 1984). Proximate cues that stimulate mating in lowland leopard frogs are not well studied (AGFD 2001). Rainfall and water temperature have been mentioned as cues for other leopard frog species in the Southwest (AGFD 2001).

## Habitat

Lowland leopard frogs occur in ponds and stream pools along water systems in desert grasslands to pinyon juniper (Platz and Frost 1984). The species occurs at elevations ranging from sea level to 1817 meters (Sredl et al. 1997b). 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 *in* AGFD 2001; Sredl and Saylor 1998 *in* AGFD 2001). Benedict (2002) detected this species occupying open water channels, higher elevation bedrock seeps, and an open cattle pond/spring in the Bill Williams Basin. Lowland leopard frogs occupied habitat in Arizona, consisting of 82% natural lotic habitats and 18% lentic habitats (primarily stock tanks) (Sredl et al. 1997a). In lotic habitats, the species is concentrated at springs, near debris piles, at heads of pools, and near deep pools associated with root masses (Jennings 1987 *in* AGFD 2001). Sartorius and Rosen (2000) document this species using filamentous algae (*Cladophora*) mats for concealment. Habitat heterogeneity in the aquatic and terrestrial environment appears to be an important factor for lowland leopard frogs (AGFD 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 hibernacula (Jennings 1987 *in* AGFD 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 semipermanent aquatic systems, this species may survive the loss of water by retreating into deep mud cracks, mammal burrows, or rock fissures (Howland et al. 1997). Recent data from the population along the Bill Williams River found that frogs favored shallow braided channels with small amounts of emergent vegetation (Cotten and Leavitt 2014).

Riparian overstory at extant lowland leopard frog localities in Arizona include cottonwoods (*Populus* spp.), willows (*Salix* spp.), baccharis (*Baccharis glutinosa*), mesquite (*Prosopis* spp.), and salt cedar (*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). Lowland leopard frog 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 Department 2004).

Populations of lowland leopard frogs do not appear to be affected by the majority of flash flood events (Sredl et al. 1997a); scouring floods may be beneficial to the populations of this species. 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 lowland leopard frogs. University of Nevada, Las Vegas, researchers conducted a habitat use study for the closely related relict leopard frog and found that relict leopard frogs strongly avoided segments of high vegetation cover, especially where *Scirpus* spp. were present (Harris 2006).

Historically along the LCR, lowland leopard frogs inhabited slackwater aquatic habitats dominated by bulrushes, cattails, and riparian grasses near or under an overstory of cottonwoods and willows. Lowland leopard frogs were also observed in canals, roadside ditches, and ponds in the Imperial Valley as desert lands were converted to agriculture (Jennings and Hayes 1994a, 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 lowland leopard frog larvae feeding on filamentous algae (*Cladophora*) mats and the organisms within them. Other species of leopard frogs of the *R. 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 the feeding behavior or diet of lowland leopard frog 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 the 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 frogloggers depend on the identification of calls made by amphibian species. Calling surveys used by Cotten and Leavitt (2014) found that lowland leopard frogs readily responded to the playing of recorded calls and were easily heard when present. 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 widely used methods that provide estimates of species richness, species using the habitat, and the proportion of habitat occupied by a 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 through 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). An accurate and precise determination of leopard frog population sizes requires the use of mark-recapture methods (Donnelly and Guyer 1994).

Sredl et al. (1997b) use mark-recapture techniques to monitor populations of lowland leopard frogs. Sartorius and Rosen (2000), Sredl et al. (1997b), and Clarkson and Rorabaugh (1989) use visual encounter surveys to monitor populations of this species. Sartorius and Rosen (2000) monitored egg masses of one lowland leopard frog population in a 2-km segment of the Agua Caliente Canyon. Frost and Platz (1983) monitored the presence/absence of egg masses for this species of the *R. pipiens* complex in the Southwestern United States. The AGFD 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 lowland leopard frog survey protocol, but visual encounter and mark-recapture surveys are the most common techniques used with ranid species.

## Threats

Lowland leopard frogs have been extirpated from more than 50% of their historical range and are believed to be extirpated from the LCR due to habitat loss, fragmentation, and introduction of non-native 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 drainage of wetlands (Clarkson and Rorabaugh 1989; Jennings and Hayes 1994b). Damming, draining, and diverting water have fragmented formerly contiguous aquatic habitat dispersal corridors necessary for establishment or maintenance of functional metapopulations (NatureServe 2006). Non-native species establishment, in particular predatory fish, crayfish (*Orconectes virilis*), and American bullfrogs (*R. catesbeiana)*, in historical lowland leopard frog habitat have been a major factor in the decline of this species (Clarkson and Rorabaugh 1989; Jennings and Hayes 1994b; Sredl et al. 1997b). Clarkson et al. (1986) conclude that the success of bullfrogs and native fishes in the LCR is the likely cause of the disappearance of lowland leopard frogs in this area. Sredl et al. (1997b) demonstrate a strong negative association between native ranids and non-native predatory fishes. Predatory fishes, bullfrogs, and crayfish block potential dispersal corridors between available aquatic habitats. The University of Nevada, Las Vegas, is conducting a study on the effects of non-native predatory fishes on another species of leopard frog (the relict leopard frog) in the LCR. The study will help determine whether introduced fishes can be removed from a section of a spring by use of fish barriers and common eradication techniques and to determine whether the removal of fishes increases egg and tadpole presence and metamorph-juvenile frog recruitment (Jaeger et al. 2004). Other factors that have contributed to the decline of lowland leopard frogs 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 salt cedar, drought, and disease (Clarkson and Rorabaugh 1989; Jennings and Hayes 1994b; Sredl et al. 1997b).

Lowland leopard frog populations in the San Felipe Creek drainage were eliminated by flooding and increased salinity levels (Jennings and Hayes 1994a, 1994b). Ruibal (1959) found that salinities greater than 5 0/00 (parts per thousand) were lethal to developing eggs in *R. pipiens* (lowland form). Salinities ranging from 3.8 to 4.6 0/00 were semilethal 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 *R. pipiens* (lowland form) is between 6 and 13 0/00 (Ruibal 1959).

The previously mentioned factors that have contributed to the decline of lowland leopard frogs disrupted the 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 non-native 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 non-native species (Sredl et al. 1997b). The low connectivity of lowland leopard frog populations suggests that this species is unlikely to recolonize sites when populations are extirpated (Goldberg et al. 2004).

The Rio Grande leopard frog (*R. berlandieri*) was introduced into the LCR near Yuma, Arizona, from either Texas or New Mexico between 1965 and 1971. Rio Grande leopard frogs were 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, Rio Grande leopard frogs have expanded their range west into Imperial Valley and south along the Rio Colorado, and they have been found just north of Imperial Dam on the Colorado River (Clarkson and Rorabaugh 1989; Rorabaugh et al. 2002; Cotten and Leavitt 2014). Rio Grande leopard frogs have 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 1994b; Platz et al. 1990; Rorabaugh et al. 2002). This species invades new habitats by dispersal via rivers, agricultural areas, ditches, and canals, and through introduction by humans (Rorabaugh et al. 2002). Rio Grande leopard frogs appear to coexist with bullfrogs and, on occasion, replace them (Jennings and Hayes 1994b). Lowland leopard frogs were eliminated from the LCR before the introduction of Rio Grande leopard frogs; therefore, Rio Grande leopard frogs have not appeared to be a factor in the extirpation of lowland leopard frogs from the LCR (Jennings and Hayes 1994b). The presence of Rio Grande leopard frogs may prevent recolonization of lowland leopard frogs along the LCR, but there are no data on the effects of Rio Grande leopard frogs on native fauna (NatureServe 2006; Platz et al. 1990). Rio Grande leopard frogs are large leopard frogs, and in Texas, the 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 lowland leopard frogs, may suffer both in terms of predation and reproductive competition from Rio Grande leopard frogs (Platz et al. 1990).

Bullfrogs are an introduced species in the Southwest that may have an effect on native leopard frog populations. During the summer of 1981, they were detected at an average density of 9.1 per linear kilometers in the LCR between Laguna and Morelos Dams, Arizona-California (Clarkson and DeVos, Jr. 1986). Bullfrogs appeared to be significant predators of lowland leopard frogs when they were declining in southeastern Arizona (New Mexico Game and Fish Department 2004). Moyle (1973) cites bullfrogs as the single most important factor in the elimination of northern red-legged frogs (*R. aurora*) from the San Joaquin Valley. Schwaibe and Rosen (1988) conclude that bullfrogs were one of the reasons why populations of lowland leopard frogs and other species of leopard frogs were decreasing on the San Bernardino National Wildlife Refuge. Hayes and Jennings (1986) conclude that existing data did not support the hypothesis that bullfrogs were the most important agent in ranid species decline and that other causes, such as non-native fishes and habitat alteration, were equally responsible. Data on the precise timing of habitat modification relative to the introduction and establishment of bullfrogs and other exotic predators are not available and, thus, cannot provide insights into which factor is most significant (Jennings and Hayes 1994b). Preliminary data from bullfrog removal experiments are inconclusive as to whether bullfrog control measures may augment recruitment in lowland leopard frogs and other species (Schwaibe and Rosen 1988). Sartorius and Rosen (2000) found that bullfrogs have appeared to replace lowland leopard frogs in modified habitats (reservoirs; large, deep stock ponds; and other impoundments) in their historical range in Arizona. *R. catesbeiana* appear to be absent in native lotic habitats where extant populations of *R. yavapaiensis* occur in central Arizona (Sartorius and Rosen 2000; Clarkson and Rorabaugh 1989).

Fire is known to have an impact on populations of lowland leopard frogs. The Box Canyon fire in Saguaro National Park caused large amounts of ash, gravel, and coarse sand to be carried to ephemeral stream channels. Within 3 years after the fire, all but a few of the 32 pools in the Loma Verde Wash, where this species 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 U.S. Geological Survey, 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 the potential sediment yield from unburned areas in the event of future uncontrolled fires (Parker 2005).

Chytridiomycosis is a cutaneous infection of wild frogs and toads caused by the fungal agent *Batrachochytrium dendrobatidis.* Chytridiomycosis was found to be the cause of death in 29 lowland leopard frogs, 2 Chiricahua leopard frogs, and 2 canyon tree frogs (*Hyla arenicolor*) collected at 8 locations in southern, central, and eastern Arizona. Frogs were collected during December 1992, October – February 1997–98, and December – February 1998–99 (Bradley et al. 2002). Lesions found on the frogs were consistent with chytridiomycosis 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 lowland leopard frogs experience 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 *R. boylii* and lowland leopard frogs was sporadic and unrelated to the dose or strain of chytrid bacteria. Die offs in the wild may be a combination of chytridiomycosis and other factors such as habitat loss, pesticides, non-native predators, drought, temperature, and/or stress (Richards 2004; Davidson et al. 2003). Outbreaks of bacterial infections, including chytridiomycosis and red-leg, can be caused by low air temperatures and overcrowding. Sredl et al. (1997a) documented two occasions in which lowland leopard frog populations were stressed due to drought and low temperatures, which brought on a bacterial infection (red-leg) that reduced the population size dramatically.

Predators of lowland leopard frog tadpoles are suspected to be insects (Belostomatids, Notonectids, Dytiscids, and Anisopterans), vertebrates (native and non-native fishes, tiger salamanders (*Ambystoma tigrinum*), and 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 non-native fishes, American bullfrogs, Sonoran mud turtles (*Kinosternon sonoriense*), garter snakes (*Thamnophis* spp.), great blue herons (*Ardea herodias*), black hawks (*Buteogallus anthracinus*), and mammals (rats, coyotes [*Canis latrans*], gray foxes [*Urocyon cinereoargenteus*], raccoons [*Procyon lotor]*, ringtail cats [*Bassariscus astutus*], coatis [*Nasua nasua*], black bears [Ursus americanus], badgers [*Taxidea taxus*], skunks [*Mephitis mephitis* and *Spilogale gracilis*], bobcats [*Felis rufus*], and mountain lions [*Puma concolor*]) (Sredl et al. 1997a). There have been no detailed research studies on predators of lowland leopard frogs (Sredl et al. 1997a). Large adults likely eat juvenile frogs or large larvae, but no research studies have been conducted (Sredl 2005).

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# Townsend’s Big-Eared Bat (*Corynorhinus townsendii*)

## LEGAL STATUS

Two eastern subspecies of Townsend’s big-eared bats (*Corynorhinus townsendii*), Ozark big-eared bats (*C. t. ingens*) and Virginia big-eared bats (*C. t. virginianus*), have been listed by the U.S. Fish and Wildlife Service (USFWS) as endangered under the Endangered Species Act (USFWS 2006a, 2006b). The Bureau of Land Management (BLM), in California, has placed Townsend’s big-eared bats on their animal sensitive species list (BLM 2004). State designations include mammalian species of special concern in California and a species of conservation priority by the Nevada Department of Wildlife (NDOW) (Williams 1986; NDOW 2005). In 2013, the California Department of Fish and Wildlife was evaluating Townsend’s big-eared bats to be under the California Endangered Species Act (Bonham 2013). The Western Bat Working Group (Western Bat Working Group 1998) lists Townsend’s big-eared bats as a species of “Red or High” priority, the highest priority available. The International Union for Conservation of Nature 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 bats 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 range of Pale Townsend’s big-eared bats (*C. t. pallescens*) included the eastern portions of those Pacific coast States and provinces, 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 the Eastern and Central United States. The central subspecies Ozark big-eared bat’s range includes southeastern Kansas, northeastern Oklahoma, northwestern Arkansas, and southwestern Missouri. The eastern subspecies Virginia big-eared bat’s 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 Townsend's big-eared bats 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 bats (Cockrum et al. 1996; Brown 2006). New evidence concerning the distribution of the western subspecies, *C. t. townsendii* and Pale Townsend's big-eared bats, has arisen. A recent deoxyribonucleic acid (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. The range of *C. t. townsendii* now encompasses not only their original area but all other areas that were formerly designated as Pale Townsend's big-eared bat 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 the LCR MSCP Planning Area

The Pale Townsend's big-eared bat was included under the Lower Colorado River Multi-Species Conservation Program (Bureau of Reclamation [Reclamation] 2004) as an evaluation species on its covered species list. Due to recent genetic analyses, the lower Colorado River (LCR) is in the range of *C. t. townsendii* rather than Pale Townsend's big-eared bats. Because of this, under the LCR MSCP, only the species name should be used and not any subspecific names. The LCR MSCP planning area includes all of the Colorado River from Separation Canyon, in the lower end of the Grand Canyon, to the Mexico border and includes full pool elevations of the three main reservoirs (Lakes Mead, Mojave, and 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 planning area (Reclamation 2004). There have been a number of historic roosting sites along the LCR. Usually, mines included along the LCR are not actually within the planning area; however, if a particular bat species' probable foraging areas are inside the area, they are considered LCR populations. Townsend's big-eared bats were 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 Townsend's big-eared bats (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 and Berry 2004).

There are two known roosting sites inside the Lake Mead National Recreation Area. 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 2005, personal communication). Townsend's big-eared bats have been recorded acoustically from March through October at the Las Vegas Wash, which empties runoff from the Las Vegas Valley into Lake Mead. Usually these bats are 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 Biological Consulting 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

The nomenclature for Townsend's big-eared bats 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) continues this nomenclature. In the 1990s, three more detailed phylogenetic and morphologic studies were performed, which all concluded that *Corynorhinus* should be given back their full generic status for North American species (Frost and Timm 1992; Tumlison and Douglas 1992; 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).

Townsend's big-eared bats are medium-sized bats with wingspans of 30–34 centimeters (cm), forearm lengths of 3.9–4.7 cm, and weights between 8–14 grams. 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 millimeters) 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 them from the four other large-eared bat species that may be found along the LCR, which 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 Pallid bats (*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 (Arizona Game and Fish Department 2003; Kays and Wilson 2002).

## Breeding

Breeding occurs in 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 winter and will store sperm until 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 the 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 lifespans 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 bats are considered Lepidopteran specialists because at least 90% of their 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, Townsend's big-eared bats take their prey in the air, although Howell (1920) notes evidence of foliage gleaning (Kunz and Martin 1982; Pierson et al. 1999). They are considered to be slow fliers and highly agile and maneuverable (Dalquest 1947; Hayward and Davis 1964; Findley et al. 1972). This species leaves their 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 decreased how often they returned to the roost; once the young mature, the females do not return until sunrise (Clark et al. 1993; Clark et al. 2002).

## Habitat

The foraging habitat varies widely between area and subspecies. The Virginia big-eared bat was found to forage more in open fields, pastures, and cliffs rather than in nearby forested areas (Sample and Whitmore 1993; Burford and Lacki 1995). The Ozark big-eared bat was 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 kilometers (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 Townsend's big-eared bats concentrated foraging activity along the edges of riparian vegetation and generally were found in the vicinity of vegetation when traveling to foraging areas from their 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. 2000b). 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.

Townsend's big-eared bats roost 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, 2000b; Fellers and Pierson 2002; Clark et al. 2002; Mazurek 2004). They can be found at a wide range of elevations from sea level to 2400 meters (m), with most records coming from around 915 m (Pierson et al. 1999; Arizona Game and Fish Department 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 Townsend’s big-eared bats. Townsend’s big-eared bats form clusters on open surfaces of roost sites 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 19 degrees Celsius (°C) in cooler areas to 30 °C 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 Townsend’s big-eared bats and Mexican big-eared bats (*C. mexicanus*) were found to only have a difference in temperature of 6 °C or less in 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 and 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 by 3.64 m, 2.42 by 3.33 m, and 2.18 by 1.97 m. The room in one of the caves measured 6.06 m high and 9.39 m wide, while the other cave was not measured due to its large size and many internal passages, but it 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, which was probably due to the fact that caves were 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 Townsend's big-eared bat maternity roosts in California included (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 height of 2.5–5.0 m
- Roost area minimum large enough for flying forays
- Light quality of semidark to dark
- Temperature of 18–30 °C
- Humidity of 19–93% (relative humidity)
- Distance to water of within 100 m for coastal populations and 8,000 m for others

Unlike maternity colonies, bachelor (and non-reproductive female) roosting sites usually contain 1 to several individuals, although 1 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; Lacki et al. 1994). Humphrey and Kunz (1976) found a maximum of 6 males in a roost together, with an average of 2 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 2005, 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, 2000b) 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 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 they are mainly used as a place to feed on large prey items that cannot be eaten in midflight. Bats generally do not 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 Townsend's big-eared bats) 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 Virginia big-eared bat in the area; this enabled the researchers to learn more about foraging habitats by identifying the type of habitat the moths generally used (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). Swarming roosts have recently been found to possibly be an important part of Townsend's big-eared bat habitat needs. These swarming sites appear to be a place for bats to prepare for hibernation. Hibernacula can be the same location as swarming roosts but not always (Ingersoll et al. 2010). It was found that these roosts had higher minimum internal temperatures than hibernacula, and both roost types had low maximum internal temperatures (Ingersoll et al. 2010).

Winter roosting sites, or hibernacula, in the Western 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; 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). Townsend's big-eared bats begin to arrive at 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, Townsend's big-eared bats 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 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, Townsend's big-eared bats select roosts with cold, stable temperatures and moderate airflow (Humphrey and Kunz 1976; Kunz and Martin 1982). Temperatures have been found to range from -2.0–13.0 °C, with temperatures below 10 °C 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 Townsend's big-eared bat 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 their 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 Townsend's big-eared bats include black rat snakes (*Elaphe obsolete*), spotted skunks (*Spilogale putorius*), house cats (*Felis catus*), ringtails (*Bassariscus astutus*), and black rats (*Rattus rattus*) (Pearson et al. 1952; Pierson et al. 1999; 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 (*Procyon lotor*), weasels (*Mustela* spp.), predatory songbirds (Passeriformes), frogs (Anura), large spiders (Araneae), and even other bats (Fenton 2001). While humans are not predators of bats, the negative image some people have about bats has caused them to harass or even harm them. (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 Townsend's big-eared bats 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 they 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).

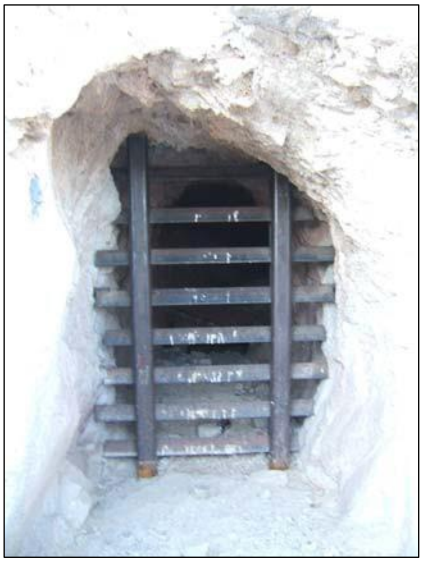

**Figure 1.—Example of a gated mine at the Salt Creek Hills Mine near Baker, California.**

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 and Kunz 1976; Kunz and Martin 1982; Pierson and Rainey 1998). Disturbances at hibernacula may also be a danger because they cause an increase in activity. This higher level of activity may cause the bats to expend too much energy, resulting in starvation (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 effects 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 (*Lymantria dispar dispar*) also tend to lower the numbers of other moth species, in turn decreasing the prey base for moth specialists such as Townsend's big-eared bats (Sample and Whitmore 1993). Conversion of native habitat to agriculture and grazing lands also threatens foraging habitat for bats. The proximity of good foraging habitat may be a determining factor in roost selection. Brown (2006) observed that Townsend's big-eared bats 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 a Townsend's big-eared bat using hollows of redwood trees in California may be important to forest management in those areas (Pierson et al. 1999; Mazurek 2004).

A recent threat to bats is white-nose syndrome (WNS), which is caused by a fungus (*Pseudogymnoascus destructans*) that grows on the face and wings of a bat while they hibernate. The fungus causes the bats to arouse more frequently, which causes them to lose fat reserves and eventually starve to death if they cannot find food. The fungus was first discovered in a cave in upstate New York after dead bats were found at a hibernaculum, and it is believed that it was originally accidentally introduced from Europe. WNS has now spread as far west as eastern Missouri and as far south as northern Alabama (Cryan et al. 2013). Currently, a total of seven species have been affected by WNS (Frick et al. 2010). To date, the two eastern subspecies of Townsend's big-eared bats have not been affected by WNS even though the fungus has been found within their range (A. Froschauer 2005, personal communication).

Threats specific to the LCR include both the disturbance of roosts and foraging habitats. 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 bat population declines along the LCR (Pierson and Rainey 1998; Brown 2006).

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