

Development of Invasive Plant Species Monitoring Protocol for Park Units in the Chihuahuan Desert Network, National Park Service

Progress Report (October 2008 – August 2009)

Submitted To:

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Introduction

The Chihuahuan Desert Inventory and Monitoring Network (CHDN) is one of thirty-two networks in the National Park Service charged with developing long-term natural resource monitoring plans for their park units. The CHDN is comprised of seven national parks, monuments, recreation areas, and historic sites in New Mexico and Texas. The CHDN identified the need for seven protocols for long-term monitoring of CHDN vital signs (Reiser et al. 2008). One of these necessary protocols is related to invasive plants.

Executive Order 13112 established the National Invasive Species Council and established a definition of invasive species for federal agencies. The Council defined an invasive species as “an alien species whose introduction does or is likely to cause economic or environmental harm or harm to human health” (U.S. Department of State 1999). The introduction, establishment, and spread of invasive species have received heightened attention because of their associated negative effects on global biotic integrity (Stohlgren et al. 1999). Likewise, the incursion of invasive plant species has become a challenge to the protection and management of resources in National Parks throughout the United States. Ecological effects depend on the rate and amount of spread, site characteristics, and the biology of organisms in infected areas. Although initial establishment of invasive plants species can result in increased biodiversity, prolonged occupancy and spread by non-native species can disrupt user interactions and communities, and ultimately lead to a less diverse biota (Radosevich 2007).

The CHDN Invasive Plants Monitoring Protocol will guide Park efforts in monitoring existing invasive plants and assist in early warning of approaching invasive plant species. The protocol will be used to monitor the distribution and spread of existing invasive plants to provide a metric for management success. Research associated with protocol development will provide information to help in understanding the introduction, spread, and distribution of invasive plant species and their effects on native habitats. Identification of distributional pathways and estimating the exposure of biotic resources to invasive species through risk assessment approaches, provide the framework for developing conservation strategies to restrict or eradicate invasive species. This effort will help prevent costly restoration and spread of new invasive species into CHDN park units. The CHDN Invasive Plants Monitoring Protocol will be designed to determine long-term trends in the distribution and rates of spread of focal invasive plants toward and within network park boundaries.

In July 2008, the Chihuahuan Desert Network Coordinator and Cooperative Unit Staff developed a scope of work to address the CHDN purpose and need to develop an invasive plant species monitoring protocol for park units in the Chihuahuan Desert Network. This scope of work outlined objectives and deliverables for NMSU and the NMCFWRU (New Mexico Cooperative Fish & Wildlife Research Unit) that would assist in meeting the purpose of need of the Network. A five-year Cooperative Ecosystem Studies Unit (CESU) agreement between NPS and NMSU was established to accomplish this work. Project objectives by phase include:

Project Objectives

1. Conduct a scoping meeting with the appropriate CHDN personnel
2. Develop an outline of an invasive plant monitoring protocol for CHDN based on approved monitoring protocol

3. Prioritize invasive species that will be the focus of predictive model development for early detection
4. Compile ecological tolerances on selected species and compile data on known plant occurrences
5. Compile images and initiate image processing (this task to be completed in Phase III, contingent upon the availability of funding), and initiate remote sensing analyses in White Sands National Monument (WHSA), and portions of BIBE (this task to be completed in Phase III, contingent upon the availability of funding)
6. Generate models and predictive maps showing the probability of occurrence of focal invasive species (to be completed in Phase III)
7. Work with the CHDN Quantitative Ecologist to develop a sampling frame for assessing the accuracy of the model predictions (i.e., the maps of predicted probabilities of occurrence) of focal species (to be completed in Phase III)
8. Assist NPS in developing a monitoring protocol for the early detection of invasive plants in the Chihuahuan Desert.

Summary of Activities

Project Kickoff Meeting:

Cooperative Research Unit staff and NPS personnel met during the vital signs kickoff meeting (November 2008) in which introductions were made and the CHDN Protocol Development Summary was reviewed. Current project objectives, monitoring goals, and basic approaches were presented and discussed in detail. Various questions and comments were addressed and formalized with representatives of all the individual park units. Comments and reviews of the scope of work were appropriately edited and incorporated into the final CHDN Protocol Development Summary document.

Monitoring Protocol:

A draft outline for the monitoring protocol using the format and content suggest by Oakley et al. (2003) was submitted for review in December 2008. This protocol uses previous protocols developed for the NPS as a template including the Sierra Nevada, Upper Columbia Basin, Rocky Mountain, Klamath, Northern Great Plains, and Heartland Networks. The importance of the protocol is to identify status and trend of invasive species on CHDN parks. This protocol provides the link between management and monitoring, builds on current monitoring funding, and provides baseline information for associated NPS programs (Fancy et al. 2009).

Species Prioritization:

Unit staff investigated an invasive plant prioritization process suitable for use in the park units located in the Chihuahuan Desert Network. We reviewed existing monitoring protocols for invasive species including the Sierra Nevada, Upper Columbia Basin, Rocky Mountain, Klamath, Northern Great Plains, and Heartland Networks. The Gulf Coast Network Vegetation

Structural monitoring protocol and the Dinosaur National Monument Invasive Plant Management Plan were also reviewed. These existing protocols have either placed certain species on network watch lists or have incorporated systems of prioritization that include either prioritizing species by management concerns or scoring species by a system of weights and ranks. We initially considered a method of weights and ranks for the CHDN; however, this system relies on numeric values that can, at times, be considered arbitrary. Given this, a technique was developed that is based directly on the evaluation of factors that affect species invasion and are considered important to early detection (Appendix 1). These factors focus on location, size of plant populations, plant invasion stages, and the likelihood of the plant to pose an ecological or management problem. The decision tree or dichotomous key is a repeatable process that minimizes individual bias, and is open to review and revision as deemed necessary.

Experts from each park are an integral part of the species prioritization process. We rely on experts to provide critical information about the plant invasion evaluation factors for their park unit. We first sent park staff a ‘master’ list of every possible invasive species in their park along with corresponding questions regarding the invasive evaluation factors. This ‘master’ list documented all possible invasive plants species in the network and was received directly from the CHDN. The master list was generated by the CHDN, which compiled herbarium records with information from past and current research projects. The invasive evaluation factors were added to the master list which addressed specific park related issues. These issues included 1) known plant locations in the park, 2) if the populations were abundant or widespread, 3) if the populations were increasing or stable, and 4) if plants pose any immediate management concerns. NMCFWRU staff reviewed these lists and park personnel were contacted to discuss data entries. These evaluation factors were then linked to the decision tree and modifications were subsequently made.

Clarification of the prioritization process and identification of the final priority species list took place during the invasive plant prioritization workshop. The workshop was held on June 23, 2009 and representatives of all the park units with the exception of Fort Davis NHS attended. During the workshop current project objectives, monitoring goals, and basic approaches were presented and materials were provided to solidify understanding of the invasion evaluation factors that lead to early detection. The workshop required individuals representing the park units to be divided into workgroups based on park proximity. These breakout groups were encouraged to discuss issues involving their individual park and to elicit park staff opinion and review of the prioritization process. The focus of the workgroups was to finalize the entries of the evaluation factors for each species recorded in their respective park. Once the entries were finalized, the individual park datasets were compiled into a network wide dataset. Species were then evaluated based on the decision tree using a SAS program. Errors in the program and in the dichotomous key were discussed with park personnel. The draft priority species list and park unit species data entries were finalized during the conclusion of the workshop.

After the workshop, the draft priority list was then further evaluated by Unit staff. Concurrently, errors in data entries (i.e. misspellings, capital words, etc.) were addressed. A total of 142 records were generated for Tier 1b, 2b, or 3b species. The majority of the species were considered Tier 6 species which describes plant occurrences that are in the county in which the

park is located but these occurrences are more than 1 mile from the park boundary. The entire data summary from the workshop can be reviewed at:

<http://fws-case-12.nmsu.edu/case/NPSinvasive/workshops.htm>

There are 15 species with 30 data records in the Tier 1a, 2a, or 3a classification (Table 1). The 30 data records represent species by park, such that individual species occur on multiple parks. Specifically, these data records include 4 species in Tier 1a, 10 species in Tier 2a, and 7 species in Tier 3a. Data records are park specific and some species were ranked in different Tiers according to the respective park. As an example, Malta starthistle was ranked as a Tier 1a species in White Sands National Monument, but listed as Tier 2a in Carlsbad Caverns National Park. These species were then further reviewed based on knowledge of life histories and the ability to create habitat specific models that would address early detection issues and management concerns of the species.

Table 1. List of 15 Priority Invasive Plant Species for Early Detection and Monitoring in Chihuahuan Desert Network. This Species List Resulted from the Prioritization Workshop between NPS and NMCFWRU.

Scientific Name	Common Name	Number of Parks Spp. Occurs
<i>Arundo donax</i>	African/Giant reed	1
<i>Bothriochloa ischaemum</i>	King Ranch bluestem	1
<i>Bromus tectorum</i>	Cheat grass	1
<i>Centaurea melitensis</i>	Malta starthistle	5
<i>Eragrostis lehmanniana</i>	Lehmann lovegrass	2
<i>Marrubium vulgare</i>	White horehound	1
<i>Peganum harmala</i>	African rue	2
<i>Pennisetum ciliare</i>	Buffelgrass	1
<i>Ricinus communis</i>	Castor beans	1
<i>Salsola</i> spp.	Russian thistle	3
<i>Sapium sebiferum</i>	Chinese tallow	1
<i>Sorghum halapense</i>	Johnsongrass	3
<i>Tamarix aphylla</i>	Athel	2
<i>Tamarix</i> spp.	Saltcedar	3
<i>Verbascum Thapsus</i>	Wooly mullein	1

Compilation of Species Life Histories:

Life history knowledge of an invasive plant species is helpful in determining the general range and environmental characteristics required for the establishment and spread of the species. Life history information can also aide in future control efforts and is important in determining appropriate spatial data to model such invasions by helping to identify input variables that generate the predicted models. Modeling endeavors that are removed from the underlying ecology of the organism may not be effective (Belovsky et al.2004). As such, we present brief species profiles for each target species (Appendix 2). We describe the origin of each species, provide a general description, and indicate vectors, pathways, and habitat associations that are useful in modeling potential distributions. We also present brief methods of control.

Species Specific Location Data:

In 2000, the Exotic Plant Management Program was created to help combat invasive plants at a national scale. Each region within the National Park system has an Exotic Plant Management Team (EPMT). The EPMT performs a variety of tasks in each park including the prevention of new introductions of exotic species, the reduction and control of existing infestations, and the restoration of native plant communities and ecosystem functions. Another task of the EPMT is to provide rapid responses and treatments to new infestations of invasive plants before they spread.

The Chihuahuan Desert and Southern Shortgrass prairie EPMT (CDSP-EPMT) treats a specific group of invasive plants in the region. These species may change slightly from year to year depending upon plant response to past treatment. CDSP-EPMT has treated several thousand acres of invasive plants with 19 species of exotics currently under management. Because many parks in the CHDN are near invasive weed dispersal pathways and are bordered by areas of degraded range and pastureland, the CHDN parks are under continuing threat for re-infestation by previously controlled species and the introduction of new invasive species. The CDSP-EPMT identified three new highly aggressive invasive species (hoary cress, diffuse knapweed, and field bindweed) and successfully treated parks for these species before becoming widely established.

An additional task of the CDSP-EPMT is to document the site of invasive plant populations and record size and locations of treated areas. Currently, the top 10 species addressed by the CDSP-EPMT include Saltcedar, Russian olive, Siberian elm, Russian thistle, Canada thistle, Malta starthistle, African rue, horehound, buffelgrass, and Johnsongrass. Because several of these species overlap with our early detection priority list, data records of known species occurrences can be used to support the habitat modeling process. NMCFWRU staff have identified and obtained existing spatial datasets including the Alien Plant Control & Monitoring Database (ApCam) from the regional NPS office, the Exotic Plant Management Team (EPMT) database, with data records spanning all park units in the network, and various other park specific spatial datasets (specifically those from Guadalupe Mountains and Big Bend National Parks). Collaboration with the CHDN and the Carlsbad Caverns GIS team is expected to help to create a more user-friendly data source for future collaborators and park staff. Additional spatial datasets collected by Unit staff are listed in Appendix III. These spatial datasets will also be used in the habitat and risk models of invasive plants.

Future Activities (Phase III – Currently Unfunded)

Knowledge of invasive species occurrence, distribution, and potential invasion pathways is important in developing appropriate long-term monitoring protocols. However, costs associated with ground-based visits preclude land managers from inventorying all areas of interest to determine invasive species presence. One potentially cost-effective approach in identifying potential occurrences of invasive species is to predict their distributions using remotely sensed data and knowledge of species ecology and environmental tolerances (Everitt et al. 1996, Goslee et al. 2003, Osborn et al. 2002, Parker-Williams and Hunt 2004). Once potential areas of invasive species occurrences are predicted, ground reconnaissance can be more effectively used and applied to an early detection context for monitoring, verification, and control.

Increases in trans-oceanic commerce, terrestrial transportation systems, and varying land use practices have allowed many plants to have either accidentally or deliberately been introduced into places that are well beyond their native range (Moody and Mack 1988). Few communities are impenetrable to invasion by nonnative species, although communities differ in their susceptibility to invasion (Sakai et al. 2001). As such, early detection and monitoring of invasive plants in the CHDN focuses on locating spatially rare events across large geographic areas, e.g., detecting new invasive plant introductions or detecting areas that are susceptible to the spread from an existing population. This type of plant detection and monitoring may preclude standard sampling designs as often general ground based searches are not efficient or effective due to plant lag times (Smith et al. 1999, Binggeli 2001).

To narrow the search frame and to be most effective with limited funds, Phase III incorporated modeling the potential distribution of selected invasive plants and conducting a risk analysis to identify areas that may have the highest likelihood on invasive plant incursion within the Network. Preliminary research on predictive modeling in Big Bend National Park occurred in 2007 (Young et al. 2007). Additional research evaluating the efficacy of using remotely sensed data for early detection of invasive plants in the Chihuahuan Desert was conducted by Young et al. (2009) on Holloman Air Force Base, and was the focus of a Ph.D. Dissertation by Young (unpublished data). Updates that utilize additional information regarding creating predictive and risk assessment models for BIBE have been completed (Young, unpubl. data) and can be incorporated into Phase III. Objectives related to Phase III that were outlined in the Scope of Work include creating and assessing invasive species predictive models, and assisting the Network in finalizing the draft monitoring protocol for submittal to the formal NPS peer review process.

Predictive modeling and associated risk analyses may not be necessary for each of the target plant species across each Park unit. Plants and areas subjected to these analyses should be discussed between Unit and Park staff.

Although Phase III is included in the Scope of Work developed by CHDN and NMCFWRU staff, this part of the project is currently unfunded. Much of the work and coordination to complete the invasive plant species monitoring protocol was under the direction of CHDN Quantitative Ecologist. Since this position was not filled, NMSU and NMCFWRU staff is posed and willing to take a larger role in assisting the Network in completing the monitoring protocol. Before NMSU and NMCFWRU dives into Phase III, we suggest we revisit the CHDN purpose and Need, and adjust the direction of the project appropriately.

Mutual Benefits:

NMSU and NMCFWRU personnel have a broad expertise with invasive plant modeling and monitoring, and have assisted other Federal and State agencies in developing monitoring strategies for conservation areas. Federal and State agencies that partner with the NMCFWRU and the Center for Applied Spatial Ecology benefit by having a variety of expertise from USGS, and a variety of expertise from departments at NMSU (e.g., invasive plant ecology, spatial modeling, experimental statistics, wildlife and range ecology, and geography) focus on management issues and problems, and customize research and technical assistance projects to address the appropriate questions. Likewise, NMSU and NMCFWRU benefits from NPS research and technical assistance. By being involved in NPS issues and culture that could lead to

the biodiversity conservation and the potential implementation future projects with the CHDN and other I&M networks or parks, and by providing students (undergraduate and graduate) and interns with opportunities and education that pertain to Chihuahuan Desert ecosystems.

Literature Cited:

- Belovsky, G.E., D.B. Botkin, T.A. Crowl, K.W. Cummins, J.F. Franklin, M.L. Hunter, A. Joern, D.B. Lindenmayer, J.A. MacMahon, C.R. Margules, and J.M. Scott. 2004. Ten suggestions to strengthen the science of ecology. *BioScience* 54:345-351.
- Binggeli, P. 2001. Time-lags between introduction, establishment and rapid spread of introduced environmental weeds. *Proceedings of the Third International Weed Science Congress*; 2000 June 6-11; Foz do Iguassu, Brazil, Manuscript number 238, 17 p., CD-ROM. Available from: International Weed.
- Everitt, J. H., D.E. Escobar, M.A. Aleniz, M R. Davis, and J.V. Richerson. 1996. Using spatial information technologies to map Chinese tamarisk (*Tamarix chinensis*) infestations. *Weed Science* 44:194-201.
- Fancy, S.G., J.E. Gross, and S.L. Carter. 2009. Monitoring the Condition of Natural Resources in US National Parks. *Environmental Monitoring and Assessment*. 151:161-174.
- Goslee, S.C., K.F. Beck, and D.P.C. Peters. 2003. Distribution of Russian knapweed in Colorado: Climate and environmental factors. *Journal of Range Management* 56:206-212.
- Moody, M.E., and R.N. Mack. 1998. Controlling the spread of plant invasions: the importance of nascent foci. *Journal of Applied Ecology* 25:1009-1021.
- Oakley, K. L., Thomas, L. P., & Fancy, S. G. 2003. Guidelines for long-term monitoring protocols. *Wildlife Society Bulletin*, 31, 1000–1003.
- Osborn, S., V. Wright, B. Walker, A. Cilimburg, and A. Perkins. 2002. Linking wilderness research and management-Volume 4. Understanding and managing invasive plants in wilderness and other natural areas: an annotated reading list. Gen. Tech. Rep. RMRS-GTR-79-Vol 4. U.S. Forest Service, Rocky Mountain Research Station, Fort Collins, CO. 65 p.
- Parker-Williams, A.E., and E.R. Hunt. 2004. Accuracy assessment for detection of leafy spurge with hyperspectral imagery. *Journal of Range Management* 57:106-112.
- Radosevich S.R. 2007. *Ecology of Weeds and Invasive Plants: Relationship to Agriculture and Natural Resource Management*, 3rd Edition. 472 pp.

- Reiser, M. H., S. L. Garman, M. A. Powell, J. T. Richie, and J. P. Ward, Jr. 2008. Chihuahuan Desert Network vital signs monitoring plan. Draft report. Natural Resource Report NPS/CHDN/NRR—2009/XXX. National Park Service, Fort Collins, Colorado.
- Strohlgren, T.J., D. Brinkley, G.W. Chong, M.A. Kalkhan, L.D. Schell, K.A. Bull, Y. Otsuki, G. Newman, M. Bashkin, and Y. Son. 1999. Exotic plant species invade hot spots of native plant diversity. *Ecological Monographs* 69:25-46.
- Sakai, A.K., F.W. Allendorf, J.S. Holt, D.M. Lodge, J. Molofsky, K.A. With, S. Baughman, R.J. Cabin, J.W. Cohen, N.C. Ellstrand, D.E. McCauley, P. O'Neil, I.M. Parker, J.N. Thompson, and S.G. Weller. 2001. The population biology of invasive species. *Annual Review of Ecology and Systematics* 32:305-332.
- Smith, H.A., W.S. Johnson, J.S. Shonkwiler, and S.R. Swanson. 1999. The implications of variable or constant expansion rates in invasive weed infestations. *Weed Science* 47:62-66.
- U.S. Department of State. 1999. Executive Order 13112. *Federal Register* 64:6183-6186.
- Young, K.E., T.S. Schrader, K.G. Boykin, C. Caldwell, and G.W. Roemer. 2007. Early Detection of Invasive Species in Big Bend National Park: Remote Sensing and GIS Strategies. New Mexico Cooperative Fish and Wildlife Research Unit. Las Cruces, New Mexico. 103 pp + appendices.
- Young, K.E., C. Caldwell, and L.B. Abbott. 2009. Early Detection of Invasive Plant Species on Holloman Air Force Base, New Mexico. New Mexico Cooperative Fish and Wildlife Research Unit. Las Cruces, New Mexico. 89 pp.

Appendix II. Draft Ecological tolerances for the priority species.

Buffelgrass (*Pennisetum ciliare*)

Origin and Range

Buffelgrass (*Pennisetum ciliare*) is native to Africa, southern Asia, Europe and the Middle East (GRIN 2000). Buffelgrass was initially planted in Texas in 1917 but it failed to become established until a second planting occurred in 1947 near San Antonio (Hanselka 1988). The grass now occurs in California, Arizona, New Mexico and Texas as well as a few eastern and Midwestern states (NPS).

Description

Buffelgrass is a warm season perennial. It grows up from 10-150 cm in height and in bunches. Its active growth period is in the summer and fall but moisture is necessary for growth to occur. As a result growth patterns may vary depending on the geographic location of the plant (Hanselka 1988). The leaves are 2-7 cm long, keeled and often reddish in the cooler months (USGS Weeds). Inflorescences are cylindrical, between 2-13 cm and 1-2.6 cm wide. They are purple, grey or yellow in color. The spikelets are surrounded by bristles.

Reproduction and Pathways

This grass can reproduce vegetatively or by seed. The seeds are typically asexually produced although sexual reproduction does sometimes occur (USGS Weeds). The seed can be spread by wind, water, animals (including humans) and vehicles. The bristles on the seeds allow it to attach to animals and vehicles as well as facilitating wind dispersal (USDA NRCS 2006). Seeds can remain viable for as long as four years (Winkworth 1971). Seedlings grow slowly and can be easily overcome and outcompeted by native plants or weeds (Bovey et al 1984).

Habitat Associations

Buffelgrass is associated with loamy soil, loam sand, sandy clay (Mutz and Scifres 1975). Moisture is a main determinant in germination. Soils heavy in sand typically do not hold enough moisture to allow the plant to survive (Hanselka 1988).

Control Methods

Buffelgrass is best controlled by the use of herbicides. For a good review of possible chemicals and the best times to use them, see USGS WEEDS and Bovey et al 1984. There are no biological control methods available at this time. Fire is generally beneficial to Buffelgrass. It regrows quickly after burning, alters fire regimes and can expand after fire has swept through (Morisawa 2000).

Appendix II Cont.

Cheat Grass (*Bromus tectorum*)

Origin and Range

Cheatgrass is introduced invasive species that is originally native to Eurasia and the Middle East (Keeley 2007). The plant first appeared in British Columbia, Washington and Utah during the period of 1889-1894 (Mack 1981). The exact method of introduction is not known but it most likely occurred as a result of contaminated feed. At this point in time cheatgrass can be found throughout the entirety of North America (NRCS 2009). In eastern states the plant is usually found along roadside or in meadows, in the west it is often the dominant plant species covering vast acreages (Kaufman 2007).

Description

Cheatgrass is an annual, winter annual or biennial grass that typically grows from 5-60 cm tall (Mitich 1999). The stems are green and thin, about 2- 4 mm thick. The panicle is long and drooping with a green or purple reddish color. The awns protruding from the seed head are long, about 12-14 mm in length (Morrow 1984). It grows in dense stands.

Reproduction and Pathways

Cheatgrass reproduces entirely by seed. The seeds typically germinate in the fall, grow through the winter and set seed in the spring but in some cases spring germination does occur (Mitich 1999, Morrow 1984). The seeds are capable of persisting for up to three years in the soil though most will last only one year (Smith et al 2008). Seeds are dispersed by wind, contaminated seed and the long awns allow them to attach to animals (Morrow 1984). The awns are stiff and capable of puncturing animal mouths, eyes and organs.

Habitat Associations

Cheat grass grows in all soils except hard packed soil, it occurs at elevation of 2000-700 feet and requires little water (Hull 1947). In the western states it occurs in large swaths of pastureland often in a nearly complete monoculture. In eastern states it can be found in pastures or disturbed areas such as road sides (Kaufman 1997).

Control Methods

Cheatgrass can be contained through grazing, chemicals or biocontrol agents. Grazing can control the population but since overgrazing facilitates invasion by both cheatgrass and other species, this might not be the best option. Chemicals can be applied either in the fall or spring. For more detailed information on chemical types and use see Colorado State Parks weed profile. For biocontrol a rhizobacterial strain is available for cheatgrass control in winter wheat (Kennedy et al 2001). It works by specifically inhibiting root formation in cheatgrass. It is not currently available for wild use. Other forms of biocontrol are under evaluation (Meyer et al 2008).

Appendix II Cont.

Chinese Tallow (*Sapium sebiferum*)

Origin and Range

Chinese tallow is native to China. It was imported into the US by Benjamin Franklin in 1776 (Kaufman 2007). The tree now occurs throughout the southern United States west to Texas and also in California (NPS).

Description

Chinese tallow is a deciduous tree capable of reaching heights up to 60 feet with a diameter of 3 feet (Kaufman 2007). The tree produces flowers in spring, fruit starting in the late summer and continuing until fall. The most distinctive traits of Chinese tallow are its leaves and fruits. The leaves are broad at the petiole and narrow to a point at the tip. They are green for most of the year but turn a striking red color in the fall. The fruits are composed of three white lobes; in each lobe is a seed. The fruit pods are said to resemble popcorn. Flowers small, yellow and produced on 20 cm long spikes (EPA).

Vectors and Pathways

Seeds are the means of dispersal for this tree. The seeds are dispersed by birds and water (Jubinsky and Anderson 1996). Chinese tallow has spread throughout the United States by ornamental plantings. In Florida it is now illegal to sell the plant. No data is available on seed longevity

Habitat Associations

Chinese tallow can be found in wet prairies, bottomland hardwood forests and other moist areas (Jones 1989). In Texas coastal prairies and flood plain forests are being replaced by this plant (Bruce 1997). The main requirement for this plant is moisture. Its spread appears to be limited by (Bruce 1997,

Control Methods

Chinese tallow can be controlled by hand pulling when the plants are young and populations are small. Trees can easily be killed by cutting but the tree will re-sprout vigorously if an herbicide is not applied. Herbicides like Garlon 4 ® can be applied to the bark or they can be applied to cut stumps (Tallow Plan). For more information on control of Chinese tallow in various situations see Tallow Plan

Appendix II Cont.

Giant Reed (*Arundo donax*)

Origin and Range

Giant reed is native to the countries surrounding the Mediterranean east to southeast Asia (Goolsby 2009). It has spread beyond this native range throughout Europe and north America due to its use as an ornamental plant. The invasion of the United States has been facilitated by its use to prevent erosion along ditches (Perdue 1958).

Description

Giant reed is a tall reed grass that is typically between 1 and 8 meters tall. The culms are from 1 to 4 cm in diameter and a yellow-green color when mature (Perdue 1958). The culms are hollow with partitions, similar to bamboo, at the nodes. The leaves emerge from the nodes. They are elongate and 5-8 cm in length with a wide base that narrows to a fine tip. The plant flowers emerge from large plume like panicles at the top of each culm during August and September. The rhizome root masses produced by a stand of giant reed can become large and tangled up to 1 m deep (Bell 1997). Giant reed is capable of incredible growth. When conditions are favorable the plant can grow as much as .7 m per day.

Reproduction and Pathways

Arundo donax does not typically spread by seed. For reasons that are currently unknown the seed produced by the plant are infertile (Perdue 1958, Bell 1997). The plant spreads into new areas vegetatively. Bits of broken stalk or rhizomes are capable of washing downstream and becoming established (Bell 1997). Once established in a location, giant reed spreads through clonal propagation of its rhizomes. Reproduction from rhizomes allows the plant to quickly recover from fire and outcompete native plants following a fire (Bell 1997).

Habitat associations

Giant reed is capable of growing on many types of soil including hard clay (Perdue 1958). In the southwest it is associated with waterways and riparian areas due to its requirement for large amount of water to allow establishment and maintain its ability to grow at an astounding rate (Perdue 1958). Though giant reed is associated with riparian areas it is capable of surviving a period of drought up to several years in duration once it has become established (Bell 1997, Perdue 1958).

Control Methods

This plant can be controlled by mechanical, chemical or potentially biological means. The product Rodeo® is potentially 100% effective at killing the plant rhizome and is certified for use near water ways (Bell 1997, Tracy and Deloach 1998). Studies are being conducted at this time to determine a suitable biological control method for this plant (Moran 2009). At the time of publication there are no biological controls available, however.

Appendix II Cont.

King Ranch Bluestem (*Bothriochloa ischaemum*)

Origin and Range

King ranch bluestem (*Bothriochloa ischaemum*) first arrived in California from Amoy, China in 1917 (Celarier 1955). By 1937 it had been discovered in the King Ranch in Texas (Celarier 1955). Since that time king ranch bluestem has been planted widely as forage. It can now be found in from California through the southwest, in several western states and through much of the southeast (NPS).

Description

King ranch bluestem is an erect bunchgrass that is capable of forming a dense lawn (Celarier 1955). Hairs present at leaf nodes. The plant grows from 18-48" tall; it is green in color and turns yellow when mature (TexNat). Spikelets are found on several branches of the panicle (TexInvasives).

Vectors and Pathways

This plant reproduces primarily from seed. Its spread appears to be due largely to intentional plantings, either for forage or erosion control along road ways (TexInvasive).

Habitat Associations

In Texas this plant can be found from the south Texas plains to the Gulf Prairie marshes (Sammon 2005). It can survive in loam, clay, sandy soils and clay loam (FAO).

Control Methods

Unknown at this time

Appendix II Cont.

Johnsongrass (*Sorghum halapense*)

Origin and Range

Johnsongrass (*Sorghum halapense*) is originally from the Mediterranean regions of Africa and Europe. It was brought to this country in the 1830's and was first introduced into South Carolina (FEIS). The intended use was as forage for livestock. The plant has now spread across the entirety of the United States with the sole exception of Maine (NPS).

Description

Johnsongrass is large perennial grass in warm weather areas. It can grow up to eight feet tall and possesses a white mid-vein in its leaves which grow up to two feet in length (Kaufman 2007). The possession of a white mid-vein is helpful in distinguishing Johnsongrass from other similar grasses. This grass produces seed in a large, up to 2ft long, purplish panicle (Kaufman 2007). The overall appearance of the plant is quite similar to corn.

Reproduction and Pathways

Johnsongrass reproduces through seeds or rhizomes. Rhizomes function to store carbohydrates as well as for the vegetative production of shoots (Anderson 1960). Seeds are produced throughout the summer and into the fall (Anderson 1960). The seed of this plant are also capable of long term viability, at least 5 years, in the soil (Egley 1983, Leguizamon 1986). Seeds can be distributed by wind, water, agricultural machinery and wildlife (Hartzler 1991, Ghera 1993).

Habitat Associations

Johnsongrass is found in disturbed areas, roadsides, cultivated fields, ditches or any other area with a moist soil (WA state, Millhollon 1969). In the southern states it grows in fields that have long been fallow (Kaufman 2007). The northward spread of this plant has been limited only by its inability to overwinter.

Control Methods

Control and elimination of Johnsongrass is made particularly difficult due to the fact that it spreads both by rhizomes and seed. As for most weeds, this plant can be controlled by pulling or hoeing when it occurs in small patches. Herbicides, like glyphosate, work well (Penn State).

Appendix II Cont.

Malta Starthistle (*Centaurea melitensis*)

Origin and Range

Malta starthistle (*Centaurea melitensis*), is native to southwestern Europe (Callaway 2003). It was introduced into California sometime in the 17th century. The first known record of its occurrence is a seed found in an adobe brick that dates back to 1797 (Hendry 1931). Malta starthistle can now be found in many of the southwestern and western states and there are some occurrence records from a few states in the Midwest and in the east (NPS).

Description

This plant grows to a typical height of less than one meter with hairy, narrow, 1-3 cm long leaves (DiTomaso and Gehrlach 2000). It is a winter annual. Small yellow flowers with long spiny bracts are produced in April and June (DiTomaso and Gehrlach 2000). Flowers are typically produced at the end of the stems but they can also be produced in the center of the basal leaves at ground level (West 2001). The flowers have the distinct appearance of most thistles and resemble those of yellow thistle.

Reproduction and pathways

Malta starthistle reproduces by seed. This plant is capable of producing as much as 60,000 seeds in a single season (DiTomaso and Gehrlach 2000). The seeds can be distributed by animals and wind (Gerhlach unpublished). Seeds can remain dormant for as long as ten years (Coconino).

Habitat Associations

This plant can be found along ditches, road sides and fallow fields (Allred and Lee 1999). The plant is wide spread in California (DiTomaso and Gehrlach 2000).

Control Methods

Hand pulling or hoeing is effective in small infestations. Tilling if done properly can control malta starthistle (Coconino). Herbicides that are effective on yellow starthistle should be effective on malta starthistle. Application of clopyralid and picloram between the months of December and April appears to provide the best results (Texas Invasives). For more detailed information on herbicides and control see the University of California Pest Management Guides.

Appendix II Cont.

Russian Thistle (*Salsola kali*)

Origin and Range

Russian thistle is native to Eurasia. It arrived in South Dakota in the late 1800's in a shipment of flax seed (Goeden 1968). It has since spread widely throughout the United States and Canada (NRCS 2009).

Description

Russian thistle is most commonly recognized in its dead form as the western icon, the tumbleweed. In life the plant forms a rounded annual shrub that is most often a strong green color with purple stripes. It is between .3 and 1 meter tall and between .3 and 1.5 meters in diameter (Howard 1992). The leaves are round and elongate with spines on the tips. Flowers are small and found at the leaf axil. The small winged seed is retained at the leaf axil. Seeds are distributed when the dead plant is separated from its root and rolls across an area.

Reproduction and pathways

Russian thistle reproduces entirely by seed. The plant skeleton separates from its root and the seeds are distributed by the rolling skeleton over a distance of up to several kilometers (Stallings 1995). Spread of existing populations usually occurs in the direction of prevailing wind for this reason. Seeds cannot germinate in hard compacted soils and are typically dependant on disturbed areas for germination (Wallace 1968). Russian thistle seeds are quickly outcompeted and overcome through crowding by native plants (Allen 1982).

Habitat Associations

Nearly all soil types can be inhabited by Russian thistle. It can be found in disturbed areas, agricultural fields, pastureland, road sides and desert areas (Young and Evans 1972).

Control Methods

Russian thistle can be controlled through the use of herbicides.

Appendix II Cont.

Saltcedar & Athel (*Tamarix* spp.)

Origin and Range

Saltcedar (*Tamarix* spp.) is a plant originally native to Eurasia. It was brought to the US in the 1820's as an ornamental and for use as a windbreak (Kaufman 2007). It has since become established in riparian areas throughout western states up to Montana and can be found as far as the east coast (NRCS). Tamarisk is less of a problem on the northern extent of its range such as along the Snake River in Idaho (Dixon 1999).

Description

Saltcedar is a deciduous tree or bush that is usually less than 6 m tall with multiple branches or trunks. The bark of the branches and the trunk(s) is dark in color sometimes appear reddish or purplish. The leaves are feather like in appearance with a scale like texture. Flowers are suspended on a panicle that extends from the tips of the branches. They are pink or white in color, small and densely packed on to the panicle. Saltcedar gets its name from the fact that its leaves resemble that of cedar and salt glands are present on the leaves. The salt glands contribute to increasing salinity in the soil either by dripping salt directly to the ground or by falling from the tree. Saltcedar often grows in dense thickets along riverbanks and is easily identified from their feathery appearance.

Reproduction and Pathways

Saltcedar reproduces through seed production. A mature saltcedar plant releases as much as 500,000 seeds in a single season (Brotherson 1987). The seeds are dispersed by wind or water. Saltcedar is also capable of spreading vegetatively by broken bits of stem and root that wash downstream. The plant is capable of re-sprouting from existing rootstock when damaged by fire or mechanical means.

Habitat Associations

In the desert, saltcedar is confined to riparian areas, flood plains, marshes, lake edges or any other area that contains significant levels of moisture. Saltcedar is not limited to any particular soil type and thrives in soil with high salinity and alkaline conditions (DiTomaso 1998).

Control Methods

Saltcedar is not negatively impacted by fire or cutting. It vigorously grows when damaged. Arsenal® is the most commonly used herbicide for killing salt cedar (Lym 2002). Rodeo® or Glypro®, both glyphosates, can be combined with Arsenal® as well (Lym 2002). Biocontrol agents are in the preliminary stages of testing. Proposed methods, like all biocontrols, are not intended to wipe the plant out but to simply control its spread. The leaf beetle (*Diorhabda elongata*) has been approved for use by the USDA (Dennison 2009).

Appendix II Cont.

Lehmann's Lovegrass (*Eragrostis lehmanniana*)

Origin and Range

Lehmann's lovegrass originates from southern Africa (Martin and Cox 1984, Anable et al. 1992), and has spread aggressively into native grass and grass-shrub lands in the southwest. Introduction in the southwest was in 1932 with a series of experiments conducted near Tucson, Arizona (Cox et al. 1988). Starting in the early 1950s, commercial seed growers produced large quantities of Lehmann's lovegrass which was planted from Texas to Arizona to prevent soil erosion and provide livestock forage (Cox et al. 1988, McClaren and Anable 1992). Lehmann's lovegrass is presently in southwestern states ranging from California to Texas (Kartesz 1999). Lehmann's lovegrass is a warm-season, perennial bunchgrass that reaches heights from 45-61 cm (1.5-2.0 ft) (Humphrey 1970).

Description

Lehmann's lovegrass is a warm-season, perennial bunchgrass that reaches heights from 45-61 cm (1.5-2.0 ft) (Humphrey 1970). Plants are mostly erect and form short basal offsets from the base of the stem at nodes. Panicles are somewhat open, and do not form

Reproduction and Pathways

Lehmann's lovegrass spread by seed and stolons. It is a prolific seed producer, and can maintain a reserve seed supply that will persist for several years in the soil and quickly take advantage of any moisture available (Uchytel 1992). Lehmann's lovegrass is also stoloniferous. Stems that come in contact with the ground may root at the nodes. Lehmann's lovegrass is very competitive, and colonizes disturbed sites rapidly (Anable et al. 1992). Seedlings emerge following summer rains on sites disturbed by burning, mowing, or grazing (Roundy et al. 1992). Highly variable moisture regimes favor establishment because a portion of seeds remain dormant even if favorable conditions exist (Abbott and Roundy 2003). This allows for subsequent establishment after first attempts have perished due to desiccation. Established plants form nearly pure stands after several growing seasons (Humphrey 1994). New invasions are limited to 6-10 m (20-33 ft) adjacent to existing stands unless seeds are transported by livestock waste or vehicle use in contaminated areas (Anable et al. 1992, Robinett 1992). Seeds can pass unharmed through livestock and this plant is a common contaminant in feed hay, therefore this plant can easily establish in livestock feeding areas (Anable et al. 1992). Roads and utility right-of-ways, livestock use areas, and fire scars are the primary pathways for spread.

Habitat Associations

Lehmann's lovegrass can establish on most soils in the southwestern U.S. during a wet summer. However, the grass dies out during the next few years unless sites exhibit sandy

or sandy loam textured soils (Cox et al. 1988). Elevations of successful Lehmann's lovegrass establishment in the southwestern U.S. range between 2,477 and 5,050 ft (775-1,540 m), with annual rainfall between 11-20 in (28-50 cm) (Cox et al. 1988). This grass does not tolerate low temperatures (Uchytíl 1992), but establishes easily in disturbed areas, but can also colonize undisturbed areas that exhibit very low natural canopy cover (Sumrall et al. 1991, Roundy et al. 1992, Ruyle et al. 1988). In the southwestern U.S., it is associated with desert shrub and semi-desert grassland communities (Uchytíl 1992).

Appendix II Cont.

Horehound (*Marrubium vulgare*)

Origin and Range

Horehound is native to northern Africa, Asia, and Europe, and is now common across most of the U.S. (Magness et al. 1971, Simon et al. 1984). This plant is a commercially produced herb used in cooking, candy, and in cough medicine (Magness et al. 1971). Horehound is an aromatic perennial herb with 25-45 cm (10-18 in) long square stems (Sievers 1930, Simonet al. 1984). Leaves are 2.5-5.0 cm (1-2 in) in length, opposite, with a densely crinkled surface, ovate to near round and resemble mint in appearance (Sievers 1930, Stritzke 1975). Stems and leaves have white woolly pubescence. Flowers are white, occur in clusters on the upper part of the stem, and have a bilabiate corolla. Horehound flowers from June to September (Sievers 1930).

Description

Horehound is an aromatic perennial herb with 25-45 cm (10-18 in) long square stems (Sievers 1930, Simonet al. 1984). Leaves are 2.5-5.0 cm (1-2 in) in length, opposite, with a densely crinkled surface, ovate to near round and resemble mint in appearance (Sievers 1930, Stritzke 1975). Stems and leaves have white woolly pubescence. Flowers are white, occur in clusters on the upper part of the stem, and have a bilabiate corolla. Horehound flowers from June to September (Sievers 1930).

Reproduction and Pathways

Little information is available on the spread of horehound. Reproduction is from seed only. Seeds have hooked burrs that are easily transported by attaching to people and animals and can remain viable in the soil for many years (Ainsworth 1999). Seed transportation also occurs from commercial and garden uses. This plant is a common contaminant in feed hay, and therefore can easily establish in livestock feeding areas. This plant species has a slow rate of growth and will rarely persist with competition from native species (Lippai et al. 1996).

Habitat Associations

Horehound can establish in poor infertile soils and is often the first colonizer of a recently eroded area (Lippai et al. 1996). Horehound has wide environmental tolerances, including broad temperature and precipitation ranges (Simon et al. 1984). Horehound occurs in disturbed sites along roadsides, stockyards, fields, pastures, near dwellings, and dry riverbanks in scattered patches (Sievers 1930).

Appendix II Cont.

African Rue (*Peganum harmala*)

Origin and Range

African rue is native to semi-arid environments in northern Africa, the Middle East, and central Asia, and has been introduced to the U.S. and Australia (Whitson et al. 2000, Mahmoudian et al. 2002). In the U.S., African rue was first introduced near Deming, New Mexico, in 1928, (Whitson et al. 2000) for the production of red dye (Parker and Reiser 1997). It has since spread throughout the west, with populations occurring in California, Oregon, Washington, Idaho, Nevada, Arizona, New Mexico, Colorado, and Texas (Parker and Reiser 1997, Natural Resources Conservation Service 2009). Skinner et al. (2000) noted that African rue was one of the 45 frequently listed noxious weeds in the continental U.S. and southern Canada.

Description

African rue (Zygophyllaceae family) is a bright green perennial desert shrub with a bushy growth habit that reaches heights around 0.3 m (1 ft) (Parker and Reiser 1997, Roche 1991). The plant has an extensive root system; with branching roots extending 4-6 m (13-20 ft) below the surface in sandy soils (Parker and Reiser 1997, Abbott et al. 2008). In southern New Mexico, African rue can exhibit a bimodal growth stage, initiating new growth in mid to late March through late June, followed by plant senescence as soils dry in early summer, and then reinitiating growth in late July and early August in response to the monsoonal rainfall (McDaniel and Duncan 2006, Abbott et al. 2007). African rue leaves are alternate, smooth, and divided deeply into narrow segments (Whitson et al. 2000). Flower production begins in mid April with fruit maturation occurring in June and July in New Mexico (Abbott et al. 2007). Flowers are white with five white petals and produce a cylindrical 2-4 celled leathery capsules that contain 45-60 dark brown seeds (Parker and Reiser 1997, Roche 1991, Whitson et al. 2000). Little is known about the ecological consequences of African rue colonization. In New Mexico, African rue forms dense monoculture stands along roadsides and disturbed areas (K. Young, personal obs), effectively reducing native biodiversity. African rue also has several drought tolerant competitive advantages over native plants in New Mexico. African rue produces a deep root system; exhibits a phenology pattern of rapid growth and reproduction; achieves a stress induced senescence and resumes growth and reproduction following favorable environmental conditions; and seedlings can tolerate, recover, and establish during periods of drought when native plants are suppressed (Abbott et al. 2006; 2007; 2008). In addition, African rue germinates earlier in spring, and may contain allelopathic chemicals that hinder or prevent other vegetation growth (Roche 1991, Parker and Reiser 1997). These competitive advantages have allowed African rue to expand into fourwing saltbush, creosote bush, and grassland communities (Roche 1991, McDaniel and Duncan 2006, Abbott et al. 2007). African rue is also toxic to cattle, sheep, and horses, as it contains lethal alkaloids (especially the seeds and roots) (Mahmoudian et al. 2002, McDaniel and Duncan 2006). However,

Appendix II Cont.

Parker and Reiser (1997) noted that poisoning is rare because the plant is extremely unpalatable.

Reproduction and Pathways

African Rue spreads primarily by seed proximate to the parent plant, dispersing greater distances via hay, seed mixture, equipment, livestock, vehicles, water, or by adhering to feet, fur, or feathers of animals (Parker and Reiser 1997). African rue can also spread vegetatively by lateral roots producing new shoots (Michelmore 1997, McDaniel and Duncan 2006). Tilling, mowing, and blading along roadsides have done little to reduce populations and have probably increased the spread (Roche 1991, Parker and Reiser 1997). Seeds may also be transported and introduced in contaminated gravel and fill materials (Abbott et al. 2008).

Habitat Associations

African rue tolerates a wide range of precipitation and temperature conditions (Abbott et al. 2007). However, it is adapted to arid environments where it can grow in a variety of soils and alkaline conditions (Parker and Reiser 1997). African rue colonizes frequently disturbed and barren areas, including roadsides, parking areas, corrals, stockyards, degraded desert rangelands, abandoned crop fields, and oil pads (Parker and Reiser 1997, McDaniel and Duncan 2006, Abbott et al. 2007).

Appendix II Cont.

Castorbeans (*Ricinus communis*)

Origin and Range

Although it is native to the Ethiopian region of tropical east Africa, the castor bean or castor plant has become naturalized in tropical and warm temperate regions throughout the world, and is becoming an increasingly abundant weed in the southwestern United States.

Description

The castor plant is a robust annual that may grow 6 to 15 feet (2-5 meters) in one season with full sunlight, heat and adequate moisture. In areas with mild, frost-free winters it may live for many years and become quite woody and tree-like. The large, palmately lobed leaves may be over 20 inches (50 cm) across and resemble a tropical aralia. There are several cultivated varieties with strikingly different foliage colorations, including black-purplish, dark red-metallic, bronze-green, maroon, bright green with white veins, and just plain green. Although it grows very rapidly with little care or insect pests and produces a mass of lush tropical foliage, its use in cultivation should be discouraged because of the extremely poisonous seeds or "beans." Flowers occur most of the year in dense terminal clusters (inflorescences), with female flowers just above the male flowers. This species is clearly monoecious, with separate male and female flowers on the same individual. There are no petals and each female flower consists of a little spiny ovary (which develops into the fruit or seed capsule), and a bright red structure with feathery branches (stigma lobes) that receives pollen from male flowers.

Vectors and Pathways

Each male flower consists of a cluster of many stamens which literally smoke as they shed pollen in a gust of wind. The spiny seed pod or capsule is composed of three sections or carpels which split apart at maturity. Each section (carpel) contains a single seed, and as the carpel dries and splits open, the seed is often ejected with considerable force. The castorbean plant grows well in soil of medium texture.

Habitat Associations

Castor plants are very common along stream banks, river beds, bottom lands, and just about any hot area where the soil is well drained and with sufficient nutrients and moisture to sustain the vigorous growth. Castorbeans grow best where temperatures remain fairly high throughout the growing season of 140 to 180 days. The soil must be able to warm up early in the spring. The seed may fail to set, however, if the temperature stays above 100°F for an extended period. The crop requires a loamy soil of medium texture. Castorbeans do well on either alkaline or acid soils, as long as the subsoil is permeable and there is good drainage. Seed will not set if soil moisture is inadequate. Castorbeans should not be planted in an area that is subject to erosion.

Appendix II Cont.

Woolly Mullein (*Verbascum thapsus*)

Origin and Range

Woolly mullein first entered the United States in Virginia during the 1700's (Kaufman 2007). It is originally from Europe and Asia. It was used as a poison in Virginia and for its medicinal properties (Kaufman 2007). Its current range encompasses all of the United States (NRCS).

Description

Woolly mullein, also known as common mullein is an annual plant with a typical life cycle of two years. In its first year it grows in a rosette of soft fluffy leaves similar in appearance to lambs ear. The leaves are 4-12 inches long and 1-5 inches wide (Kaufman 2007). In its second year, if it has grown large enough, it sends of a large flower stalk that grows from four to ten feet in height (Kaufman 2007). The flowers are small and yellow. The plant produces a large taproot as well.

Reproduction and Pathways

Woolly mullein reproduces by seed. The seed can remain viable for an extremely long time. 700 year old seeds found in archeological digs have been proven viable (Kaufman 2007). The plant is a weak competitor, it is easily pushed out by native plants (Kaufman 2007).

Habitat Associations

Due to its weak ability to compete with native plants, woolly mullein is found only in disturbed areas. It is commonly found on roadsides, fallow fields, recently burned areas and pastures (Kaufman 2007). It needs only 3-6 inches of water per year to survive (Kaufman 2007).

Control Methods

This plant can be controlled by pulling out individual plants or the can be hoed. If a larger area is affected then glyphosate or triclopyr can be sprayed in early spring (Kaufman 2007).

Appendix III. Spatial datasets available for the Chihuahuan Desert Network.

Spatial Datasets for CHDN

All Park Specific Data from NPS GIS Data store
All current NPS Boundaries
County Boundaries
Federal Boundaries
State Boundaries
Country Boundaries
Wilderness Boundaries
Protected Area Boundaries, US
Public Land Survey System
Omernik Level III Ecosystems US
Bailey's Ecoregions US
Generalized Soils (STATSGO)
Generalized Geologic map
Historic Features
Full GNIS for NM & TX
National Hydrography Data
Vegetation growth average 1990 - 2005
Vegetation growth peak 1990 - 2005
Average Precipitation 1961-1990
Average Precipitation 1971-2000
Average Max Temp 1971-2000
Average Min Temp 1971-2000
NAIP imagery
Digital Raster Graphics (DRG's) (topo)
ETM 30M spectral bands
ETM 15M pan band
ETM 60M low and high thermal
MSS triplicate (70s, 80s, 90s)
DOQQ's
Landsat 5 TM Imagery
National Elevation Dataset 10M & 30M
2001 NLCD, canopy cover
2001 NLCD, impervious
2001 NLCD, Landover
1992 NLCD, US
Detailed roads (Bureau of Transportation Statistics)
Biomass of US forests
Forest type of US
