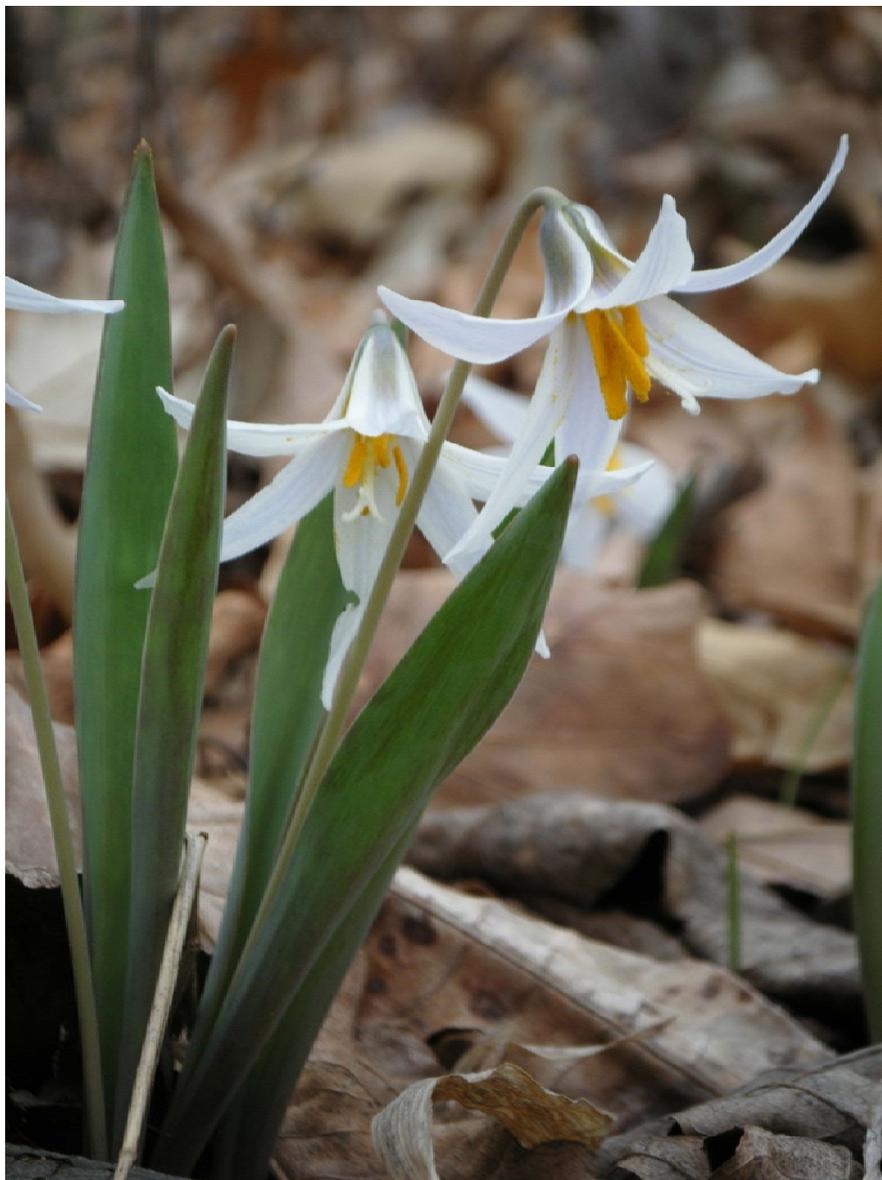


Oklahoma
Native Plant Record



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Volume 12

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Foreword

This year's *Oklahoma Native Plant Record* is all about learning from history. Publishing Dr. Marilyn Semtner's 1972 Master's thesis this year offers us an opportunity to gain another perspective on why some introduced species become invasive in natural habitats where others do not. As the Oklahoma Invasive Plant Council, formed in 2008, seeks ways to guide our state agencies to determine best practices for preserving our native plant species, research like this, both old and new, can inform policy decisions.

Mr. Randall Ledford has collected extensive information regarding use of Oklahoma's native plant species by the Pawnee Native Americans. He gives us a preliminary plant list that is sure to become part of an important resource that can be used in Pawnee cultural education and by ethnobotanists. With full respect for the Pawnee culture, his list includes scientific names as well as Pawnee names and descriptions of uses that have been carefully researched and whose content has been approved by the Pawnee elders. We are hoping to build interest and anticipation in this area of social and botanical research overlap. His goal is to collect and organize a larger body of this little known ethnobotany for wider dissemination.

Dr. Gloria Caddell and Ms. Kristi Rice have provided us with the long anticipated flora of Alabaster Caverns State Park. It also compares flora in those Gypsum outcrops with two other previous studies done ten years ago at Selman Living Lab in Woodward County and on a ranch in Major County.

Teaching and inspiring botany students at McLoud High School and at the University of Oklahoma Biological Station, Dr. Bruce Smith has contributed several articles to the *Record* in the past. This year he offers us a comparison study of two oak forests based on data collected by his students. Engaging his students in plant distribution and ecological studies, he strives to fulfill our need to collect and preserve data for the future — a future history, to be used by future scientists.

This year our "Critics' Choice Essay" is from Dr. Wayne Elisens. He tells us how new software and digitization methods are bringing new light to historical collections, virtually. Herbaria are making specimen data and images globally accessible. We will be able to see and learn from historical and current collections from all over the world.

The Oklahoma Native Plant Record will keep passing on the science and building on what we know. We do not want to lose, or fail to learn, what future generations will need to know to keep Oklahoma's native plant species thriving in Oklahoma. As our practice of publishing historical, unpublished work shows, we believe in the importance of historical studies and how they can inform current science policies and future research. Moving into the future, all previous volumes of *The Record* are now available on the internet at <http://ojs.library.okstate.edu/osu/index.php/index>, and it is listed on the Directory of Open Access Journals through <http://www.doaj.org>.

Sheila Strawn
Managing Editor

POSSIBLE MECHANISMS OF THE EXCLUSION OF JOHNSON GRASS BY TALL GRASS PRAIRIES

Submitted to the Faculty of the Graduate College of Oklahoma State University
in partial fulfillment of the requirements for the Degree of Master of Science
May 1972

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Keywords: Johnsongrass, exclusion, control, prairies, allelopathy

ABSTRACT

Historically, plant distribution typically has been studied with the purpose of learning why a species grows and survives where it does; but why a species does not survive in a particular habitat has rarely been studied, although it may be just as important. According to the US Department of Agriculture, Johnsongrass [*Sorghum halepense* (L.) Pers.; formerly Johnson grass] is listed as an agricultural pest in most states south of the 42nd parallel. Control of Johnsongrass in agricultural fields involves various labor intensive cultural, mechanical, and chemical means. Release of a bio-control agent has not been suitable for intensively cropped areas. An agriculturally important weed and prominent member of early stage secondary succession, Johnsongrass is not present in later stages of prairie succession. Various environmental factors (biotic and abiotic) that might be involved in restricting Johnsongrass survival were examined in this research. In two sites in Oklahoma, soil conditions were found to be more favorable for survival and growth of Johnsongrass in undisturbed prairie than in the disturbed areas in which Johnsongrass was found vigorously growing. However, even when its rhizomes were introduced into mature prairie, Johnsongrass did not thrive. In laboratory and field trials, presence of the living dominant prairie grasses or leachate from living or dead leaf blades seemed to influence growth and survival of Johnsongrass rhizomes. The prairie grasses, little bluestem [*Scizachyrium scoparium* (Michx.) Nash] and Indian grass [*Sorghastrum nutans* (L.) Nash], seem to play a similar allelopathic role in restricting the growth of Johnsongrass to outside of the prairies. Looking at this past study might lead to new methods for the future. (Semtner 2012)

INTRODUCTION

Plant distribution has typically been studied with the intent of discovering why a species grows where it does. Early studies of Johnson grass [*Sorghum halepense* (L.) Pers.; currently Johnsongrass] took this approach. Introduced about 1830 from Turkey, Johnson grass has vigorously and rapidly spread from the Atlantic coast to central Texas and has been recently reported in low wet places in California (Munz 1963). It is known as a sun-adapted grass that grows well at high temperatures (Ahlgren 1956).

Although it has some value as forage, it has been and is regarded as a serious weed. Adapted to a variety of habitats, Johnson grass was reported to be an aggressive invader of such disturbed habitats as abandoned and cultivated fields and roadsides, as well as rich alluvial river bottoms. Producing large tenacious rhizomes, it is extremely difficult to eradicate. Due to its invasion of cultivated fields, many attempts have been made to control it, especially by chemical means. Control methods were directed mostly

toward destruction of the rhizomes. Workers in chemical control have included Leonard and Harris (1952), McWhorter (1961), Nester (1967), Hicks and Fletchell (1967), Wiese (1968), Millhollon (1970), and Kleifeld (1970).

Secondary succession occurs in abandoned fields and other places where the vegetation is damaged or destroyed. Those plants appearing first give way and are replaced by other species. Ultimately the climax or stable vegetation consists of species that replace themselves when their life span ends. Booth (1941) divided secondary succession in old fields in central Oklahoma into 4 stages, based on species present: (1) weeds, (2) annual grasses, (3) perennial bunch grasses, and (4) climax prairie. He surveyed the vegetation present in the annual grass and bunch grass stages. No mention was made of finding Johnson grass in either of those stages. Abdul-Wahab and Rice (1967) considered Johnson grass a prominent member of the weedy stage and definitely absent from the later stages. Their observations, however, were probably made under quite different circumstances than Booth's (1941). Observations made during the current study indicate that Johnson grass flourishes in disturbed roadsides. In continually disturbed roadsides, succession seemed to be arrested in the weedy stage.

Betz and Cole (1969) noted that undisturbed native prairie resisted invasion of both weeds and woody plants. Weaver (1968) indicated that prairies were virtually closed communities with neither a great wave of immigration nor emigration. Invaders were excluded. Invasion by weeds and/or woody plants has been considered a sign of disturbance by Clements and Shelford (1939), Petty and Jackson (1966), Weaver (1968), and Black, Chen, and Brown (1969). The lack of weed and tree invasion of undisturbed prairies generally has been credited to interactions of environmental factors, abiotic and biotic, that maintain the prairie community. The more common

reasons given were climate, moisture, soil, temperature, life form and competition [fire – Vogl (1964), water – Hylander (1966), soil and water – Weaver (1968), climate and water – Grossman, Louise and Hamelot (1969), moisture and fire – Sears (1969), no one main factor by multi-influences – Costello (1969), fire and climate – deLaubenfels (1970), and climate and drainage – Vesey-Fitz Gerald (1970)]. Despite widespread observation of and comment upon the failure-of-invasion phenomenon, it has been studied very little in its own right.

My observations indicated that Johnson grass was neither an invader nor a component of undisturbed prairies, yet it might be abundant a few centimeters away in a disturbed roadside. Causes of this apparent exclusion of Johnson grass by the undisturbed prairie were unknown and unstudied. The aim of my research was to explore various possible mechanisms of the exclusion of Johnson grass by tall grass prairies.

Many factors might be involved in the exclusion of Johnson grass from undisturbed prairies. The latitude probably was influential in limiting the original spread of Johnson grass across the countryside. Wheeler and Hill (1957) reported that Johnson grass grew abundantly in the vicinity of prairies in North America, south of latitude 40°, under a wide range of climatic conditions. Ahlgren (1956) reported that Johnson grass grew vigorously as a perennial, south of the 35th parallel, from the Atlantic Coast to central Texas. Further northward, winter killing occurred. At the latitude of central Oklahoma, 36°, Johnson grass behaves as a perennial grass. Hull (1970) found that the rhizomes exhibited little or no cold hardiness at any time of the life cycle. The rhizomes were intolerant of freezing temperatures and were killed. Johnson grass, therefore, presumably was restricted from northern prairies due to the severity of the winters.

Southern prairies are subject to high summer temperatures with periods of low rainfall. Beal (1887) reported Johnson grass as an aggressive perennial grass able to withstand great heat and severe drought. Standing water was found to kill it. Ahlgren (1956) felt that abundant moisture, supplied by rainfall, stream overflow, or irrigation was beneficial but not essential for growth of Johnson grass. The climate of southern prairies generally would not be restrictive to growth of Johnson grass.

Grasses and grass communities tend to monopolize the ground against intruders. Hylander (1966) felt that grasses pre-empted living space by producing rhizomes and stolons. Tiller production dominated the surrounding area and discouraged intrusion of weeds. Weaver (1968) felt that any reproduction, spread, or establishment of weeds in prairies would need to be vegetative through rhizomes or tillers. The network of prairie plants' roots and rhizomes in the soil was so dense that "foreign" seedlings could not become established. The spread of Johnson grass by rhizome initiation has been well documented by many researchers. Hitchcock (1922) reported that Johnson grass propagated readily by seed and strong rhizomes. Anderson, Appleby, and Wescloh (1960) showed that rhizome initiation occurred 4 to 5 weeks following seedling emergence and was well developed after 6 to 7 weeks. McWhorter (1961) found that plants grown from seed produced 212 feet of rhizomes in 152 days of growth. Evans (1964) reported that rhizome growth in many grasses occurred only under long day conditions. With Johnson grass, both flowering and rhizome growth can occur together. Johnson grass flowering was accelerated by short days.

Competition for some necessary resource such as light, water, or nutrients has been commonly supposed to help the prairie resist invaders. Clements and Shelford (1939) reported that, in enclosures,

annual grasses steadily disappeared under competition by perennial grasses. Black et al. (1969) measured the efficiency of carbon assimilation in many species and concluded that more efficient species were better competitors than less efficient ones. He proposed that permanent pastures lacked weed problems because the efficient perennial grasses did not allow less efficient weeds to establish. He found Johnson grass to be an efficient species. Abdul-Wahab and Rice (1967) said that Johnson grass had excellent abilities to compete for light, minerals, and water.

The concept that one plant can influence the growth of another is well known. Competition for some necessary resource is but one such influence. Another type of influence is allelopathy, which involves chemical substances released from one plant that harms another. Substances potentially involved in allelopathy may be liberated from plants by (a) leaching of foliage by rain, (b) volatilization from foliage, (c) leaching from fallen material, and (d) root exudation (Tukey 1969). Risser (1969), in a review of competitive relationships among plants, concluded that plant interactions due to allelopathy should be separated from competition.

Pickering (1917) stated that the formation of toxins by one plant that have harmful effects on other plants or on itself was a common phenomenon. Benedict (1941) showed that dried roots of bromegrass (*Bromus inermis* Leyss.) were inhibitory to the growth of bromegrass seedlings. A sod-bound condition resulted, due to the inhibition, with vigorous growth on the edges and stunted growth in the center of a stand of bromegrass. Bonner (1950) felt that numerous species, as yet unstudied, may produce substances toxic to one or more species, and that associations or non-associations of species due to production of chemical compounds might not be uncommon occurrences. Cooper and Stoesz (1931) found that *Helianthus*

pauciflorus Nutt. (= *H. rigidus*) had an autotoxic action which inhibited or retarded growth of its own seedlings within the center of a stand. Vigorous individuals were confined to the periphery. Curtis and Cottam (1950) reported that the antibiotic and autotoxic effects of *H. pauciflorus* were due to a substance derived from decomposition of old rhizomes. They felt that, based on preliminary observations, *Antennaria parlinii* Fernald (= *A. fallax*), *Eurybia macrophylla* (L.) Cass. (= *Aster macrophyllus*), and *Erigeron pulchellus* Michx. might produce similar acting substances.

Muller (1966) suggested that allelopathy could be a significant factor in plant succession of many kinds of vegetation. Muller et al. (1964) showed that the distribution pattern of annual grassland species in Santa Barbara County, California, was influenced by volatile growth inhibitors produced by *Salvia leucophylla* Greene. In 1966, he reported that several aromatic shrubs of southern California produced phytotoxic terpenes which inhibited establishment of seedlings of a wide variety of species some distance from the shrubs. Further evidence of the toxic suppression of herb understory growth by shrubs was given by Muller et al. (1968).

Booth (1941), in his work on secondary succession in central Oklahoma, reported that the weed stage lasted only 2-3 years and that the climax grasses required 30 years or more to reinvade. Both the shortness of the weedy stage and the slow invasion by climax grasses are puzzling. Rice, Penfound, and Rohrbaugh (1960) tried to account for the slow return of climax grasses in abandoned fields by rate of seed dispersal and mineral nutrition. The rate of succession could not fully be explained by seed dispersal and mineral nutrition. Rice (1964) found widespread occurrence of inhibition of nitrogen-fixing and nitrifying bacteria by many weedy species including Johnson grass. As a result of this inhibition, a lower nitrogen level was maintained in the soil.

Parenti and Rice (1969) concluded that the first (weedy) stage was rapidly replaced by *Aristida oligantha* Michx. because several of the important pioneer species such as *Helianthus annuus* L., *Sorghum halepense*, and *Chamaesyce maculata* (L.) Small (= *Euphorbia supina*) produced toxins inhibitory to seedlings of many species of the first stage but not to *A. oligantha*. Several species of stage one eliminated species of that stage by chemical inhibition. *A. oligantha* invaded next because it was not inhibited by the substances toxic to pioneer species and was able to grow in soil too low in minerals to support species later in succession. *A. oligantha* was found to produce substances inhibitory to nitrogen-fixing and nitrifying bacteria (Rice 1964). This inhibition probably caused the longer persistence of the annual grass stage. The species of the perennial bunch grasses have higher nitrogen requirements (Rice et al. 1960).

The influence of prairie mulch or litter has not been extensively investigated. Weaver and Fitzpatrick (1934) reported that accumulations of mulch retarded growth in the spring. The soil warmed more slowly with the mulch due to reduced insolation. Weaver and Rowland (1952) experimented with growth of tall grass prairie species with and without the presence of prairie mulch. They found that the prairie with heavy litter cover had little to no understory growth. The prairie grasses that produced the litter grew better themselves with removal of the thick build-up of litter. The grasses involved included little bluestem and Indian grass. They felt the mulch was suffocating the plants. The lack of understory was attributed to the weight of the litter and decreased light being detrimental to seedling development. The seedlings would lack enough food reserve, unless they had large seeds, to grow through and above the litter. No reason was given for the limited growth of rhizomes or tillers by dominant grasses. Friend (1966) and Mitchell (1953a, b) showed that low light intensity decreased

tiller numbers in ryegrass (*Lolium* L. spp). Vogl and Bjusted (1968) and Ehrenreich and Aikman (1963) concluded that litter build-up in undisturbed prairies caused lower soil temperatures, delayed growth in the spring, and reduced yields of little bluestem, big bluestem, and Indian grass.

Muenschler (1939) reported a number of species of wild and cultivated plants to be capable of producing hydrocyanic acid, also called prussic acid, a highly poisonous substance. Johnson grass was one of many cyanogenic plants. Huffman, Cathy, and Humphrey (1963) and Kingsburg (1965) reported Johnson grass to be a pest of cultivated fields with an undesirable characteristic of forming cyanide in certain stages of development. Abdul-Wahab and Rice (1967) showed that Johnson grass produced several chemicals inhibitory to other plants that resulted in pure stands of Johnson grass by the inhibition of other early invaders of abandoned fields. The chemicals were isolated and identified. The chemicals were found to have no or little affect on plant species that occur later in succession. Substances inhibitory to nitrogen-fixing and nitrifying bacteria were also produced (Rice 1964).

Some plants have been reported that influence the presence and/or growth of Johnson grass. Penfound, Jennison, and Shed (1965) reported the replacement of a Johnson grass population by a vine-forb community. An increase of climbing bean [*Strophostyles helvola* (L.) Elliott], an herbaceous, leguminous vine, occurred at the expense of Johnson grass. They concluded that climbing bean destroyed Johnson grass by climbing up the flowering culms, weighing them down, and preventing growth by shading. Bennett and Merwine (1964) found that planting legumes with Johnson grass would enhance growth of the latter for the first 2 years due to increased fertility and nitrogen in the soil. White clover (*Trifolium repens* L.), however, offered more "competition" to Johnson grass

establishment and no gain resulted. Wheeler and Hill (1957) recommended sowing legumes with Johnson grass, if desired, for pasture. The legumes checked the tendency of Johnson grass to become sod-bound. Hitchcock (1922) reported that to utilize a Johnson grass-infested field, alfalfa should be sown. He felt that alfalfa would smother out most of the Johnson grass.

Recently, a few cases have been reported where the presence or absence of prairie grasses determined the presence of other species. Odum (1971) and Harper (1964a) concluded that the distribution and abundance of a species can be modified by the presence of associated species. Sagar and Harper (1961) showed that the presence and nature of grass communities played an important role in determining the presence or absence of weedy *Plantago* L. spp. and in limiting the size of the *Plantago* population. The *Plantago* spp. did not occur naturally within the grass community but would grow if the grasses were removed through some disturbance. Putwain and Harper (1970) concluded from their work that the grasses were mainly responsible for limiting the population size of the sorrels (*Rumex acetosa* L. and *R. acetosella* L.).

In my search for possible mechanisms of the exclusion of Johnson grass by an undisturbed prairie, various possibilities were suggested. The determining influence might be abiotic or biotic. Therefore, physical factors which might differ between the undisturbed prairie and a Johnson grass stand were explored. Many aspects of the soil were tested, including organic matter, texture, water content, and water retention ability. The effect of shading on Johnson grass growth was studied. The possibility that the prairie grasses were influencing the growth of Johnson grass was also examined. Both field and laboratory studies were utilized in an effort to determine the source of the exclusion of Johnson grass by an undisturbed, tall grass prairie.

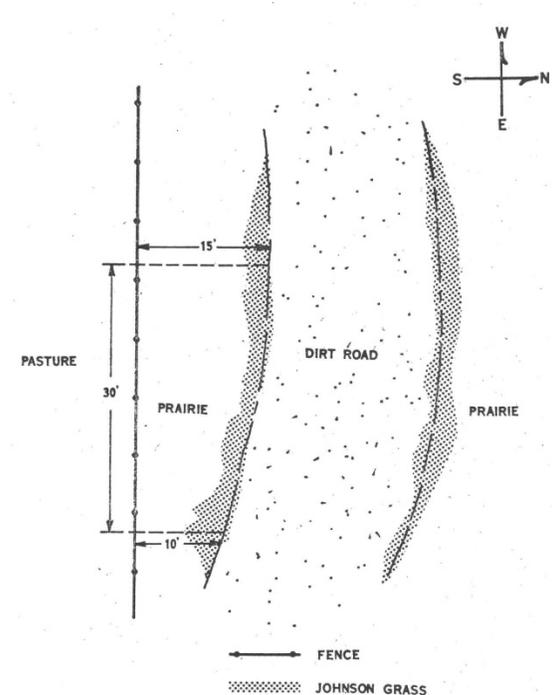


Figure 1 Blackwell Field Site near Lake Carl Blackwell, Payne County, Oklahoma

DESCRIPTION OF FIELD SITES

Two field sites were chosen in western Payne County, Oklahoma. Each consisted of a stand of Johnson grass adjacent to a prairie in good condition.

Blackwell Site

The first site was $\frac{1}{2}$ mile south of Lake Carl Blackwell. From here on, this site will be referred to as the Blackwell site. Solid stands of Johnson grass grew abundantly in the shallow ditches along both sides of a dirt road. The ditches were made some years ago and recently had been only slightly disturbed. The road was frequently graded, so Johnson grass was continually found re-invading the road from the edge (Figure 1). Although Johnson grass was continually spreading into the roadway, no spread was evident into the prairie on the opposite side.

Due to a curvature of the dirt road away from a fence, a small stand of prairie was protected from grazing. This protected area had been grazed previously, but was recovering well at the time of the study. The most prominent grasses were little bluestem [*Schizachyrium scoparium* (Michx) (= *Andropogon scoparius*)], Indian grass [*Sorghastrum nutans* (L.) Nash], silver bluestem [*Bothriochloa saccharoides* (Sw.) (= *A. saccharoides*)], and brome (*Bromus* L. spp.). Also present were small numbers of forbs, especially ones belonging to the Leguminosae and Compositae.

Preserve Site

A second site on the Oklahoma State University Ecology Preserve was selected. From here on, this site will be referred to as the Preserve site. The Preserve is located 9 miles west of Stillwater, Oklahoma, on the south side of State Highway 51 and is about 2 miles southwest of the Blackwell site. The relative placement of Johnson grass and prairie and causes were similar to those of the Blackwell site. This site was later partially destroyed by road maintenance work. The prairie within the Preserve, which remained undamaged, was used in field experiments described later.

METHODS AND MATERIALS

Soil Analysis

Soils may be responsible for vegetative distribution patterns. The exclusion of Johnson grass from undisturbed prairies could be influenced by soil characteristics. Various physical properties of the soil were explored to try to detect differences between the prairie soil and the Johnson grass soil.

Organic Matter

Organic matter (OM) was measured as an indicator of disturbance. The assumption was that the lower the OM, the more

disturbance the soil had experienced. OM was used to determine whether the soils in which Johnson grass and the prairie plants grew could be classified as disturbed. Johnson grass is usually associated with disturbed habitats.

Soil samples were taken from both the Blackwell and Preserve sites. Samples from the Blackwell site consisted of one from within a stand of Johnson grass and one from within the prairie. Samples from the Preserve were from 2 different areas within the prairie, differing in the amount of plant litter present.

Similar procedures were used to collect all the soil samples. A shovel was used to remove living plants off the surface and scrape off the top 2 cm of litter and soil. Samples were collected from the approximately 2-22 cm soil depth and consisted of pooled soil from 3 such pits. The soil was placed in appropriately labeled cardboard boxes and removed to the laboratory. After the soil was air dried in the Agronomy Department soil drying room for 24 hours, it was sieved through a #10 sieve. The OM analysis was done by the Soil and Water Service Laboratory of the Agronomy Department at Oklahoma State University.

pH

Determination of soil pH was made using a Corning Research pH meter (model 12) with equal parts by weight of air dry soil and distilled water. Soil samples were collected as previously described. Three replications were run with each soil type.

Particle Density

The particle densities were found using a pycnometer, following procedures described by Black (1965). Soil samples were collected as previously described and three replications were run.

Soil Texture

A mechanical analysis of soil was conducted to determine the percentage of

sand, silt, and clay particles. The hydrometer method as described by American Society for Testing and Materials (1964) was followed. Soil from a depth of 2-22 cm, collected as previously described, was used, as that was the region that most new roots and rhizomes occurred. Three replications of both soil types were analyzed.

Soil Moisture

Plant growth is influenced greatly by the amount of soil moisture present. During June and July 1970, soil moisture was determined regularly to detect any differences in soil moisture between the prairie and the Johnson grass stand. Soil moisture was measured by the gravimetric method (American Society for Testing and Material 1958). Soil core samples were taken during June and July 1970 from the 2-22 cm soil depth. Three transects of samples were made at the Preserve site and 5 at the Blackwell site. The transects ran from the Johnson grass stand into the prairie. Three cores were taken in the Johnson grass stand and 2 in the prairie per transect. The top 2 cm of the soil core were discarded. The remainder of the core was divided into 2 parts, 2-12 cm and 12-22 cm depth. These segments were immediately placed in aluminum cans, sealed, and returned to the laboratory.

Soil-Water Content under Different Tensions

The amount of water retained by soils at a specific pressure was measured using a porous membrane, as described by Black (1965). Soil-water contents at pressures of 0.1, 0.5, 1, 10, and 15 bars were measured. Disturbed, air-dry soil was used with 2 replications per tension, per soil type. Johnson grass and prairie soils were collected as previously described from the Blackwell site.

Plant Material

Whenever living plants were needed for experiments, Johnson grass rhizomes were collected along the dirt road adjacent to the Blackwell site. McWhorter (1961) found that plants from rhizomes grew more rapidly than plants from seeds. Hull (1970) did not detect any natural dormancy in single node rhizome pieces harvested at any time of the year. Hence, rhizomes were collected fresh as needed. Due to poor germination of local Johnson grass seeds, only rhizomes were used in the experiments.

Rhizomes were dug and placed in plastic bags. Field collected rhizomes were cut with clippers into segments containing one node. The soil in which the Johnson grass rhizomes were growing was very sandy and was easily brushed off the rhizome pieces. Rhizomes were used as soon after collection as possible.

Experiments

Seed Germination

Tests were run to determine the germination percentage of local Johnson grass seeds to decide the feasibility of using seeds as well as rhizomes in future experiments.

Seeds were collected several times from the areas of both field sites in 1970 and 1971. Germination tests were conducted with fresh and after-ripened (6-month and 1-year) seeds. Several tests were conducted according to procedures given by Tester and McCormick (1969) with 5 replications of 10 seeds per treatment. Johnson grass seeds, fresh and 6 months after-ripened, were (a) pre-chilled for 5 days at 10° C, (b) pre-chilled for 7 days at 10° C, or (c) left at room temperature. Incubation was in the dark at room temperature. The experiment was subsequently repeated with 3 variations: (a) treated with 5 percent Clorox and rinsed thoroughly with several rinses of distilled water, (b) soaked in tap water for 5 days

before pre-chilling, and (c) not treated. Germination was checked daily. A total of 450 seeds were used.

Taylorson and McWhorter's (1969) pre-chilling experiment was also tried. The procedure was to expose the seeds to 2 weeks at 10° C followed by 2 hours of 35° C and germination at 20° C in darkness. Fresh, 6-month, and 1-year-old after-ripened seeds were used with 5 replications of 10 seeds pre-treatment, for a total of 150 seeds. Germination was recorded daily.

Germination tests were also run with fresh and 6-month-old after-ripened seeds in soil from within a prairie and a Johnson grass stand. The soil was collected and prepared as previously described. Commercial river sand was used as a control. Each soil type was placed in separate Petri plates. Twenty seeds were used per replication and there were 3 replications per soil type. Tap water was used to keep the soil moist. Germination was at 20° C in the dark. The objective of the experiment was to determine whether soil type influenced germination of Johnson grass seeds.

Soil Preference in a Laboratory Situation

Soils were collected from within a prairie and a Johnson grass stand near the Blackwell site, and Johnson grass planted in them to determine whether the growth of its rhizomes might be influenced by soil type. The vegetation, litter, and top 2 cm of soil were removed with a shovel. Soil was dug up from the 2-22 cm depth and placed in standard nursery flats lined with newspaper. The soil was sieved to remove any plant parts, rhizomes, roots, etc. Flats of commercial river sand were used as controls. Three replications of each substrate with 50 rhizome pieces per flat were made on February 19, 1971.

All flats were regularly tap watered, and the number of new plants emerging and total emergence per flat were recorded every other day for 41 days. No dry weights were

taken because the plants in the soil from the Johnson grass stand were damaged by disease near the end of the experiment. A statistical analysis was made of the emergence data to determine whether Johnson grass emerged differently in any soil type relative to the others.

Growth in Disturbed and Undisturbed Field Plots

Field growth of Johnson grass from rhizomes was studied to determine if it would grow and survive in the prairie if manually planted. Rhizomes were planted under 2 conditions: disturbed (modified) and undisturbed (natural). In the disturbed plots, a 23 cm cube of soil was dug up, turned, mixed, and sieved to remove any plants and litter present. Any neighboring prairie plants that might lean over the plot were trimmed back. Five rhizome segments were planted per plot. Rhizome segments were placed approximately 4-6 cm deep.

In the non-disturbed plots, simple slits, 6 cm deep, were made in the ground with a shovel. One rhizome segment was planted in each of 5 slits per plot. No plants or litter were removed. Care was taken to avoid disturbance as much as possible. In each of the plots, the 5 rhizome pieces came from 2 or 3 different rhizomes. The procedure was repeated in a Johnson grass stand and prairie at the Blackwell site and in the prairie at the Preserve. Due to the smaller size of the Blackwell site, only 4 replications of each treatment were made in the prairie and 2 in the Johnson grass stand. Plot locations were randomized.

Eight replications were made of each treatment with 2 replications per treatment on each of the 4 transects in the prairie at the Preserve. Alternating the treatments among the subplots, each transect contained 4 subplots, 150 cm apart. Transect #1 was made in a section of the Preserve prairie that was similar to that of the prairie in the Blackwell site. In both, grass litter was light. Open spaces existed between plants where

bare soil could occasionally be seen. Along transects #2-4, deeper within the Preserve prairie, tall grass prairie was in good condition. Tall, thick stands of Indian grass and little bluestem were growing. Plants were close together with a thick layer of litter on the ground. No bare ground could be seen.

A total of 140 rhizome segments were planted. Soil at planting was moist. Soil temperatures were within a range of 13-26° C at the 7.5 cm depth and 14-22° C at 15 cm depth. This was slightly below the optimal 30° C for the maximum growth of the dominant prairie grasses and Johnson grass but well within the range for good growth. All planting was done on May 10, 1971.

Observations were made weekly to determine emergence and survival of Johnson grass. All surviving plants were harvested on September 20, 1971, and dry weights determined. Due to the extremely low numbers of plants recovered in September, no statistical analysis was conducted.

Interference Experiment

Many ecology textbooks and papers contain statements to the effect that weeds cannot compete with prairie plants. This has generally been accepted as the reason many possible invaders were excluded from the prairies. The assumption was that weeds were not efficient or successful in competing for some resource (light, water, or minerals) against the prairie plants. This statement is questionable in the case of Johnson grass. Johnson grass reportedly had excellent ability to compete for light, water, and minerals (Abdul-Wahab and Rice 1967). Black et al. (1969) showed both the dominant prairie grasses and Johnson grass to be efficient CO₂ fixing species and concluded that both were good competitors.

One resource that plants generally compete for is light. A box experiment was conducted to determine the effect of 6

different conditions. These were (1) control – full sunlight, (2) light shading – 70 percent of full sunlight obtained by 2 layers of white cheese cloth, (3) medium shading – 60 percent of full sunlight obtained by 6

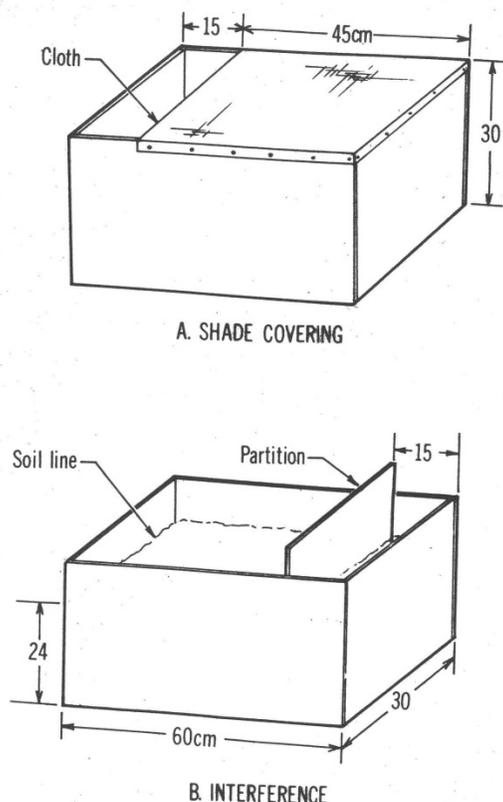


Figure 2 Box designs for the interference experiments

layers, (4) heavy shading – 18 percent of full sunlight by a tightly woven cotton cloth, (5) litter mulching – 18 percent of full sunlight with prairie litter, and (6) aerial influence with prairie grasses. A light meter was used to measure the light intensity in the field at ground level to determine the amount of shading used in the boxes. In field measurements, prairies with heavy build-ups of litter had light values down to 2 percent of full sunlight, though amounts this low were not used in any experiment.

Wood boxes were built, each 30 x 60 cm x 30 cm deep, in which the experimental plants were grown. Drainage slits were left in the bottom. The soil used was a ratio of 2 parts nursery soil and 1 part commercial sand. The cloth covers were stretched across $\frac{3}{4}$ of the boxes, approximately 6 cm above the soil level (Figure 2). Five Johnson grass rhizome segments, from 2 or more different rhizomes, were planted per box, under the shaded areas.

Prairie litter from the Ecology Preserve was collected in January, 1971 and stored in large paper bags in the laboratory until used. The litter was laid on top of the soil in the experimental boxes in amounts similar to those found in a healthy tall grass prairie with a normal build-up of litter. The litter was leached with tap water on the boxes twice weekly for a month before the rhizomes were planted.

Prairie plants were collected from the Blackwell area by randomly digging up intact clumps of prairie vegetation. Mainly, little bluestem and Indian grass were collected while dormant in early March, 1971. The clumps of prairie plants were planted in the large ends of 3 boxes and allowed to become established (see Figure 2). The previously described dirt-sand mixture was used to fill in around the prairie plants and the empty small ends. A partition was placed in the soil to divide the roots and prevent prairie plant roots from becoming established in the smaller section. After the Johnson grass plants in the smaller section had emerged, the partition was removed to allow the roots to intermingle.

The boxes were kept outdoors and were positioned in a completely randomized block design. All plants were subject to the same temperature and wind. The boxes were regularly watered. Three replications per treatment were made. The rhizome segments were planted August 25, 1971 and allowed to grow until September 30, 1971. Dry weight per plant was determined. A statistical analysis, using a hierarchical design

to compare average dry weight per plant per treatment was performed.

Effect of Plant Leachate on Growth

The hypothesis was proposed that the prairie grasses might be producing some substance inhibiting the growth of Johnson grass. It was possible that the green leaves were producing and releasing the substance, or that release was upon the death of the leaf blade. Hence, 2 separate leachates were made: (1) fresh green leaves and inflorescences of little bluestem and Indian grass, and (2) old prairie litter. In nature, any leaching would be passive due to falling rain, dew, etc., so the leaves were leached in distilled water without any grinding. Plant material was leached by soaking with distilled water for 1 hour at a ratio of 10 g of plant material per 100 ml of distilled water. The leachate was made fresh as needed, every 6 to 8 days. Leachate was stored in the dark at room temperature for periods not longer than 3 days.

Commercial river sand was used to fill standard nursery flats. Four replications per treatment with 50 Johnson grass rhizome segments per flat were planted on September 20, 1971. The flats were arranged in a partial random block design in the greenhouse. Each flat was watered with approximately 800 ml of leachate per week until October 19, 1971. For the remainder of the experiment until November 10, 1971, the plants were watered with tap water. The experiment was continued with tap water to determine if any effect on growth due to the leachate was permanent or temporary. The height of the individual plants after 29 days was recorded. The emergence per flat was recorded for 51 days.

RESULTS AND DISCUSSION

Soil Analysis

Several factors of the soil were examined to determine if these might be

responsible for the exclusion of Johnson grass by the prairie.

Organic Matter

Organic matter (OM) was tested as an indicator of disturbance. Soils sampled from the prairie had consistently and considerably higher levels of OM than the Johnson grass soil (Table I). The higher OM levels in the prairies would make the prairie soil more favorable to plant growth and root development. There is no reason to doubt that the organic matter level present in prairies would encourage Johnson grass growth rather than restrict it.

pH

Some plants are known to grow better in acidic or alkaline soils. Distribution of these species is influenced by soil pH. Johnson grass, with its wide distribution, would not seem to be greatly influenced by the soil pH. To determine if prairie soil pH was different from and thus possibly detrimental to Johnson grass growth, the soil pH of the prairie and Johnson grass sites was tested (see Table I). No significant pH differences were found. Soil pH would not be considered a factor restricting the growth of Johnson grass.

Particle Density

The particle density was determined mainly as a reference due to its influence on soil mass (see Table I). The difference between the 2 soil types was not enough to affect the soil texture greatly. The small differences in particle density would not be influential in determining the distribution of Johnson grass.

Soil Texture

Johnson grass has been reported to thrive in fine sandy loam and not grow well in deep sandy soils (Archer and Bunch 1953). The prairie soil did not appear to be a deep sandy soil, but texture analysis was performed (Table II). The prairie soil had

more silt and slightly more clay, but less sand than the disturbed Johnson grass soil. Physically, the prairie soil would appear to

favor the growth of Johnson grass more than the disturbed soil it occupies.

Table I Characteristics of 2 different soils at the 2-22 cm depth

Location	Soil area	OM ^a %	pH ^b	Particle ^c density	Litter covering
Blackwell	Johnson grass stand	0.5	6.2	2.54	Very little
	Prairie	2.8	6.0	2.45	Light to medium
Preserve	Prairie transect #1	2.5	6.2		Light to medium
	Prairie transect #2-4	3.1	6.1		Thick

^aOrganic matter, no replications
^b3 replications
^c3 replications

Table II Soil particle size analysis for 2-22 cm depth at the Blackwell site

Soil source	Percentage				Soil type
	Rep.	Sand	Silt	Clay	
Johnson grass stand	1	75	9	16	Sandy loam
	2	81	6	13	
	3	79	6	15	
Prairie	1	69	14	17	Sandy loam to Sandy clay loam
	2	60	21	19	
	3	63	19	18	

Soil Moisture

Although the precipitation received by the prairie and the roadside, separated only by a few centimeters, was similar, differences in soil moisture might occur. Considerable variation existed between samples within each soil type, separated by a few centimeters. The variation among

samples was great enough so that no large differences could be detected between soil types (Table III, Figure 3). The small differences in the soil moisture in June and July between the prairie soil and disturbed soil would not be enough to account for the presence or absence of Johnson grass.

Table III Average soil moisture in prairie and Johnson grass soils at 2 depths in 1970

Location	Date	Level	Percent moisture	
			Johnson grass	Prairie
Blackwell	June 9	T	13.0	16.5
		L	14.7	13.2
	June 16	T	10.3	9.6
		L	12.1	11.5
	June 23	T	13.7	13.2
		L	10.4	9.7
	June 30	T	6.2	7.4
		L	8.3	7.3
	July 7	T	3.6	5.0
		L	6.4	5.3
	July 21	T	13.1	14.4
		L	13.5	12.2
	July 28	T	9.0	9.1
		L	9.4	8.9
Preserve	June 11	T	12.8	15.1
		L	13.0	12.6
	June 25	T	12.0	12.6
		L	12.0	11.3
	July 9	T	6.4	4.1
		L	7.5	4.8
July 21	T	12.3	13.0	
		L	12.5	11.6

T = top soil, 2-12 cm

L = lower soil, 12-22 cm

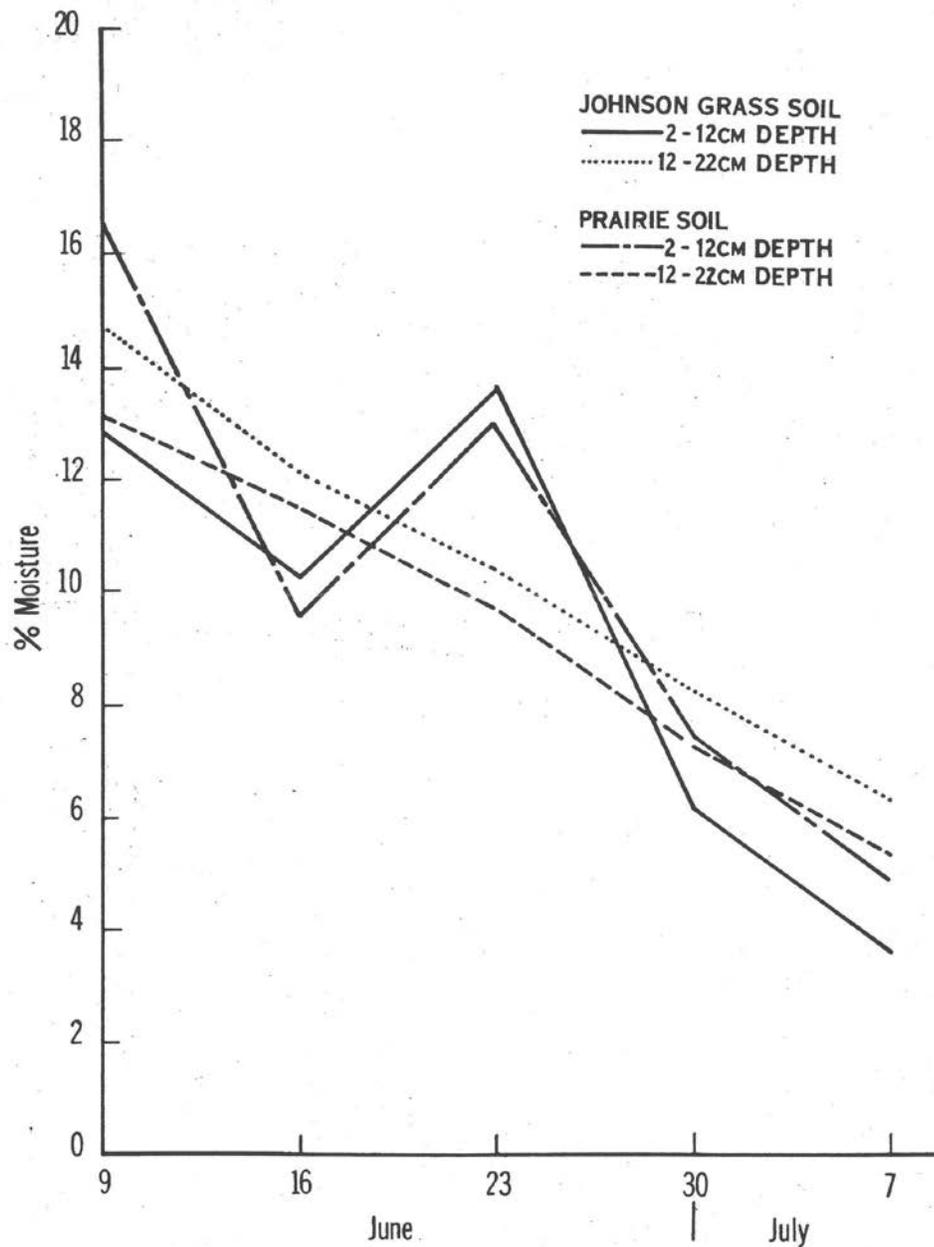


Figure 3 Average soil moisture by weight of 2 different soils at the 2-12 cm and 12-22 cm levels in June and July, 1970 at the Blackwell site.

Soil-Water Content under Different Tensions

The prairie soil held more water at any given tension than the Johnson grass soil (Figure 4). This would be expected because it has more clay, silt, and organic matter than the disturbed Johnson grass soil. Plants would have to exert more energy at any given soil-water content to obtain water

from the prairie soil compared to the Johnson grass soil. Conversely, at any given soil tension, the prairie soil would have more water available for use.

Since air-dried, disturbed soils were used, the actual values found for the soil moisture per soil pressure are not the same as would occur in the undisturbed soil profile.

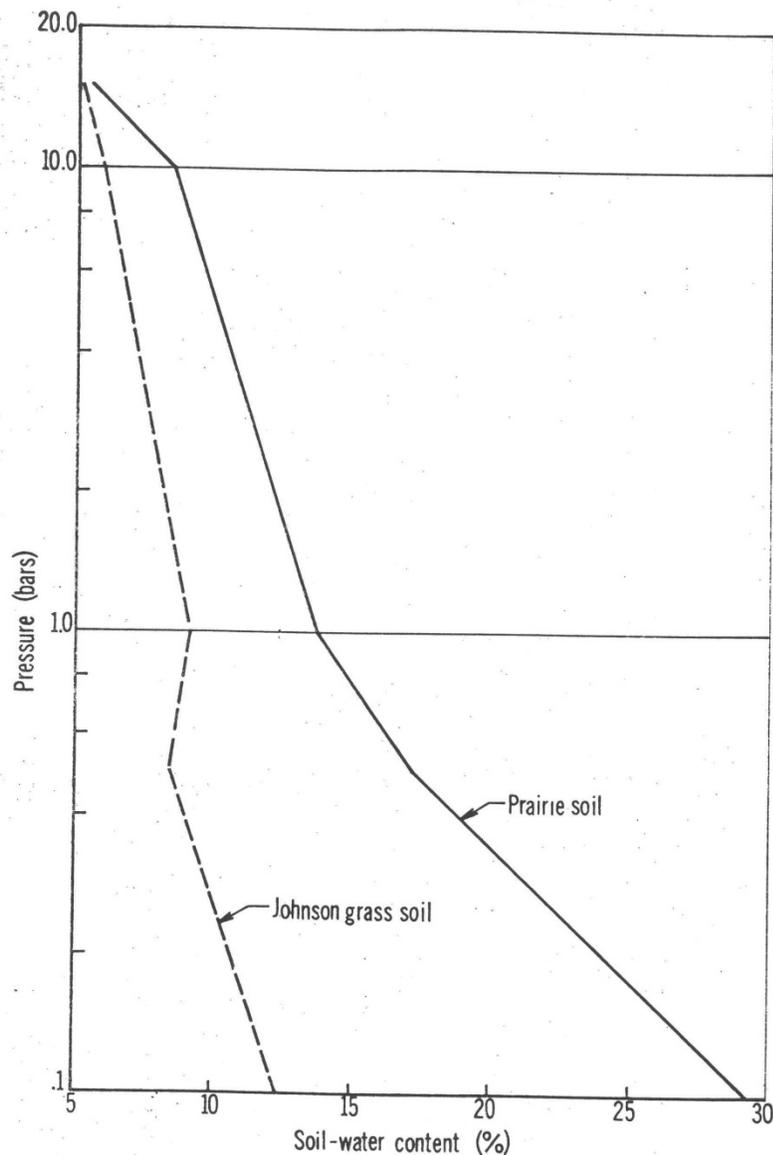


Figure 4 Soil-water content retained by air-dried soils, under different tensions (two replications per soil type).

Other Factors

Both field sites were subjected to the same climate, wind, temperatures, and rainfall. Factors were not tested if they were believed to either favor the growth of Johnson grass over the prairie grasses or to exhibit no difference between the two habitats. Rice, Penfound, and Rohrbaugh (1960) reported that the nitrogen level of the soil influenced the rate of succession. Species later in succession (*Andropogon* and *Sorghastrum*) have a higher nitrogen requirement than plants earlier in succession. As both *Schizachyrium scoparium* and *Sorghastrum nutans* were present in the prairie studied, the nitrogen probably would be higher than in the disturbed habitat. Johnson grass was known to grow better in fertile soils with high nitrogen levels (Archer and Bunch 1953, Bennett and Merwine 1964). Huffman et al. (1963) stated that Johnson grass grew on roadsides, but more abundantly where soils were of better than average fertility. The higher nitrogen levels in the tall grass prairies, compared with soils earlier in succession, would actually be beneficial to growth of Johnson grass. Logically, nitrogen levels of the prairie soil would not restrict but encourage Johnson grass growth.

Experiments

Seed Germination

Despite many different methods to try to induce germination, no locally collected Johnson grass seeds germinated in any test. Other workers have found the seeds of Johnson grass to be highly dormant (Weir 1950, Anderson 1968, Taylorson and McWhorter 1969). No seeds were used in any later trials. Seeds from local Johnson grass populations probably require a long after-ripening period.

Soil Preference in a Laboratory Situation

Initially, fewer plants emerged in the prairie soil than in the other soils, sand and

disturbed Johnson grass soil (Figure 5). This trend was not statistically significant, but was present in all replications. After the initial 2 weeks, the number of plants per flat was consistently higher in the prairie soil than in the others. The difference in the average total plant emergence after 41 days between the prairie and disturbed soil was significant only at the 20 percent level with a t-test. No significant difference was found between sand (control) and the disturbed habitat soil. Visibly, plants grown in the prairie soil were greener and taller than in the other two treatments. The increased vigor was likely due to the higher fertility of the prairie soil.

Growth in Disturbed and Undisturbed Field Plots

Study of Johnson grass planted in the field under 2 types of conditions revealed a difference in emergence and growth. In the undisturbed or natural plots, 70 rhizomes were planted, with 60 in the prairie and 10 in the Johnson grass stand. No plants emerged (Table IV). Of the 70 rhizome segments planted in the disturbed or modified plots, in the same proportions given above, 5 were alive at the end of the summer: 3 in the Johnson grass stand and 2 in the prairie. The 3 plants in the Johnson grass stand were divided between the two replications. One plant emerged shortly after planting, while emergence was delayed almost a month in the case of the other two. The cause of the difference in emergence time was unknown, but noticeable differences were seen in the dry weight and number of new rhizome segments. In the prairie, 4 plants actually emerged, in the same replication, but only 2 survived the summer.

In the disturbed sites, with all plants and litter removed, the soil was exposed to increased radiation. This produced greater heating and drying than in a comparable soil

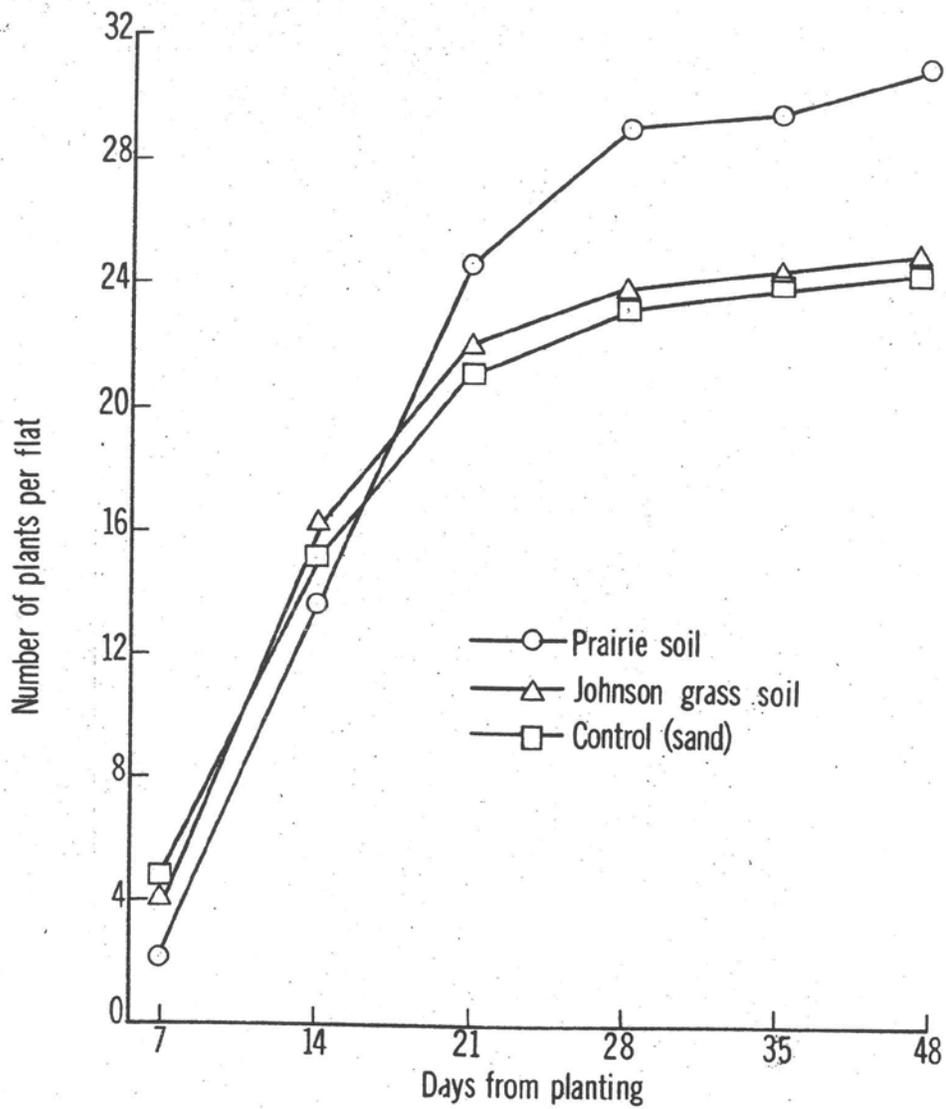


Figure 5 Effect of soil type on emergence of Johnson grass from rhizomes in flats in the greenhouse (three replications per soil type).

Table IV Comparison of field grown Johnson grass plants in 2 areas after one summer of growth from rhizome segments (May-September 1971)

Soil	Treatment	Rep.	Survival/ planted	Percent survived	Plot #	Dry wt. (g)	New rhizome nodes
<u>Blackwell</u>							
Johnson grass	Natural	2	0/10	0		-	-
	Modified*	2	3/10	30	1	1.12	7
							1.30
					2	7.10	29
Prairie	Natural	4	0/20	0		-	-
	Modified*	4	0/20	0		-	-
<u>Preserve</u>							
Prairie	Natural	8	0/40	0		-	-
	Modified*	8	2/40	6.7	1	0.04	0
						0.35	0

*Vegetation removed, soil sieved

surface protected by layers of litter and plants. A crust formed over the surface in both the Blackwell prairie plots and on the plots in transect #1 in the Preserve. These two areas were the harshest places in the experiment for Johnson grass to grow. Yet it was only in the Preserve prairie, transect #1 that Johnson grass even emerged in a prairie. In the other 3 transects, disturbed plots were soon shaded by nearby rapidly growing prairie grasses. The soil was shaded, cooler, and retained more moisture.

The number of plants emerging within a prairie and a Johnson grass stand were similar, but differences in size, dry weight, and number of new rhizome nodes were striking (see Table IV). Those in the Johnson grass stand were visibly taller, greener, and seemed healthier than those in the prairie. Those in the prairie were stunted and had yellowish foliage. In the prairie, the

plants had no new rhizome initiation, while those in the Johnson grass stand were actively producing new rhizome nodal segments.

Johnson grass growth was greatly enhanced by disturbance of the prairie soil and removal of the vegetation. The Johnson grass plants in the prairie were so stunted that survival for much longer was doubtful. Few roots were found on observation and those were very small. The reduced food storage would reduce the chances of establishment. A limited growth of Johnson grass in the prairie was obtained with removal of grasses in the immediate area.

This experiment was handicapped by not being initiated until May. During May, the soil temperatures were approaching 30° C, improving the soil temperature for growth compared with cooler soil temperatures earlier in the year. However,

the plants had very little time to develop a root system before the hot summer conditions arrived, which probably resulted in the low survival observed.

Interference Experiment

A box experiment was conducted to compare growth and emergence of Johnson grass under different conditions. In the control boxes, conditions for growth would not seem optimal. Soil was directly exposed to the sun. Heating and drying of the soil surface formed a hard crust over the soil surface. The crust served to conserve soil moisture, but also made it harder for the plants to penetrate. Growth did not seem to be restricted, as the average dry weight was higher than most of the other treatments (Table V, Figure 6). Emergence was higher than in any other treatment.

The light shade provided better conditions for Johnson grass growth. The soil retained more moisture, and less hardening of the surface occurred than in the control. Overall, those plants were the tallest and most vigorous. The thin cloth was not a barrier restricting growth. Most plants grew up through the cloth.

The cloth in the medium shade treatment was a minor barrier restricting growth in height. In two replications, the tips of a few blades reached the cover and were bent. In replication #2, the plants pushed off the cover and grew vigorously in the increased sunlight. If the average dry weight was found for the medium shading without the one strikingly different replication, the average dry weight would only be 0.3 g per plant. This would make it similar to the average values in the dark and litter treatments (see Figure 6).

Emergence was low under the deep shade, perhaps due to decreased light or temperature. The few plants that appeared were small. The growth rate was slow. None grew tall enough for the solid cloth to act as a physical barrier during the short period of the experiment. The greatly decreased light

intensity seemed to have a definite slowing effect on growth. Ryle (1967) found that ryegrass responded to shading with slower growth. Some growth of Johnson grass was obtained in all three shading treatments. Fewer plants grew with greatly decreased light, as would be found at the soil surface of prairies with heavy build-up of litter. Light was important, but would not prevent growth of Johnson grass within a prairie.

The leached litter produced shade as well as mulching and possible chemical effects. The soil remained more moist than in any but the deep shade treatment. The plants appeared above the soil surface in the boxes with the leached litter cover over a week later than in the other treatments. Variation in appearance was evident. Of the 15 rhizomes planted, 11 plants grew. A few plants appeared green and healthy, although they seemed to be growing more slowly than those in the control or with light shading. The majority of the plants were yellow-green in color and appeared stunted or at least growth was retarded. The plants emerged above the soil surface but little additional growth occurred. Two plants were thin or etiolated. Simple reduction in light intensity may explain the etiolated condition, but would not satisfactorily explain the stunting and discoloration of the Johnson grass plants under the litter. The "weight" of the litter did not prevent the plants from growing, as suggested by Weaver and Rowland (1952). Tips of a few plants were appearing above the litter. The old litter seemed to retard growth, but not prevent it.

Johnson grass plants in aerial contact with the prairie grasses were smaller with slower growth than the control or light shade treatment. The plants seemed stunted. Digging up the soil after the experiment showed no root invasion by one into the area of the other. The Johnson grass plants that did grow were greenish-yellow.

Table V Dry weight in grams and emergence of Johnson grass plants grown for 35 days from rhizomes

	Control			Light shade			Medium shade			Deep shade			Litter mulch			Competition		
	1	2	3	1	2	3	1	2	3	1	2	3	1	2	3	1	2	3
Individual weights	1.9	0.2	0.2	1.1	0.4	0.5	0.4	0.6	0.1	0.3	0.0	0.1	0.6	0.3	0.3	0.05	0.1	0.7
	0.9	0.4	0.5	0.9	1.2			1.1	0.3			0.4	0.4	0.9	0.2	0.5	1.0	
	0.2	0.9	0.5	2.3	0.6			2.1	0.1					0.4	0.1	0.45		
	0.6		0.9		0.7				0.4					0.4	0.2			
	0.5														0.2			
Means	0.8	0.5	0.5	1.4	0.7	0.5	0.4	1.3	0.2	0.3	0.0	0.2	0.5	0.5	0.2	0.3	0.6	0.7
Grand means		0.6			0.9			0.6			0.3			0.4			0.5	
Emergence percentage																		
Means		80			53			53			20			73			40	

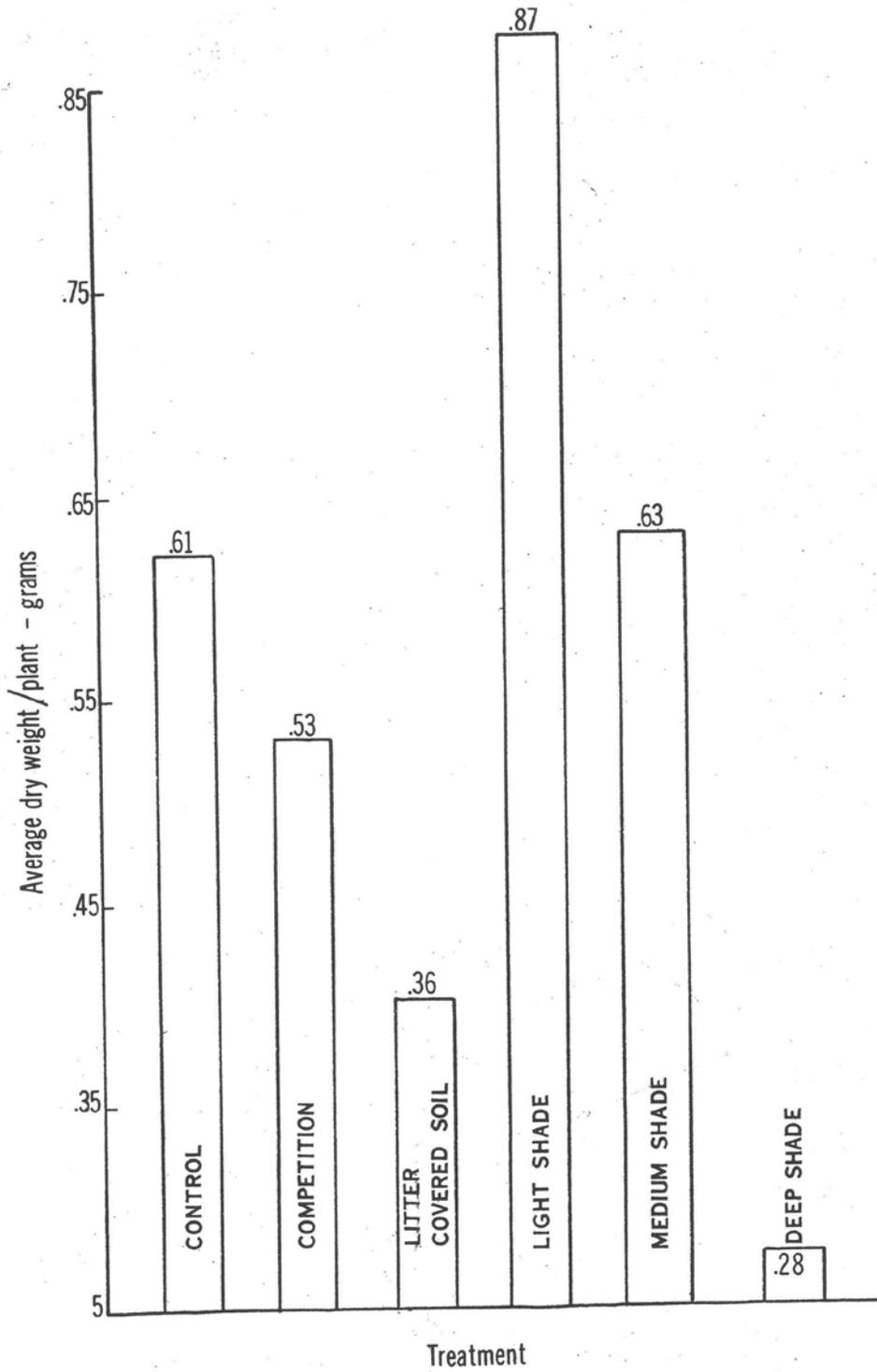


Figure 6 Dry weights of Johnson grass plants grown from rhizomes (three replications per condition).

One rhizome produced several new segments laterally in the direction away from the prairie grasses before emergence at the edge of the box. Why the rhizome grew away from the prairie grass side was unknown. After appearance above ground, little increase in height was recorded. Most of the dry weight was due to the formation of new rhizome segments rather than leaves. No new rhizome segments were produced laterally in any other replication or treatment. Without the additional weight due to the new rhizome segments on that one plant, the average dry weight in the competition boxes would be lower and closer to the average dry weight in the litter treatment. The presence of the prairie grasses within a few centimeters seemed to have as much affect as did medium and deep shading, though the Johnson grass plants were still fully exposed to the sun.

The difference in average dry weight per treatment proved significant at an 0.01 level with an F-test. Variation within treatments was evident, with the few replications used. Fluctuation in percent of emergence between treatments was not statistically significant in any reasonable confidence range due to the variation within treatments. More replications would be necessary to establish any differences in emergence between treatments.

Effect of Plant Leachate on Growth

In the two treatments watered with a leachate, fewer plants emerged, the size of the plants was smaller, and increase in height was slower than in the controls watered with distilled water (Figures 7, 8). No difference was detected between the effects of the two types of litter leachates. Those plants watered with distilled water grew more vigorously than in the other treatments. The experiment was continued after the watering with leachate was stopped to determine if the rhizomes were killed or inhibited. When the leachate was no longer

applied, many new plants appeared. An increased growth rate was evident.

SUMMARY AND CONCLUSION

Johnson grass (*Sorghum halepense*) grows abundantly in disturbed areas south of latitude 40°. In this area, it grows in disturbed roadsides and disturbed fields: beside, but not in, tall grass prairies. Johnson grass was usually growing in areas where prairie plants had been disturbed or destroyed, as along roadsides. Many stands of Johnson grass along roadsides were areas of frequent disturbances. Soil differences between the prairie and the Johnson grass stands seemed to be the result of disturbances, not natural differences. The prairie soils had a slightly different ratio of particle size and texture. The soil pH and particle densities were similar. However, the prairie soils had considerably more organic matter and were able to retain more soil moisture at any one soil tension than in the other soil. Rice, Penfound, and Rohrbaugh (1960) found that prairies with species later in succession had higher nitrogen levels than soils with vegetation of the weed stage.

Archer and Bunch (1953) reported that Johnson grass grew well on fine sandy loams, but did not thrive on poor depleted or deep sandy soils. Huffman et al. (1963) reported Johnson grass abundant on roadsides and open areas where soils were of better than average fertility. Based on physical characteristics of the two soils, the prairie would seem more favorable to Johnson grass growth than the disturbed habitat in which it grows. In laboratory tests, Johnson grass grew better in the prairie soil than in its own soil. The prairie soil did not inhibit or limit Johnson grass growth.

In the field, other factors influenced Johnson grass growth. In nature, Johnson grass grew in disturbed sites and not in the prairies. Growth was obtained in a prairie only with disturbance and removal of prairie

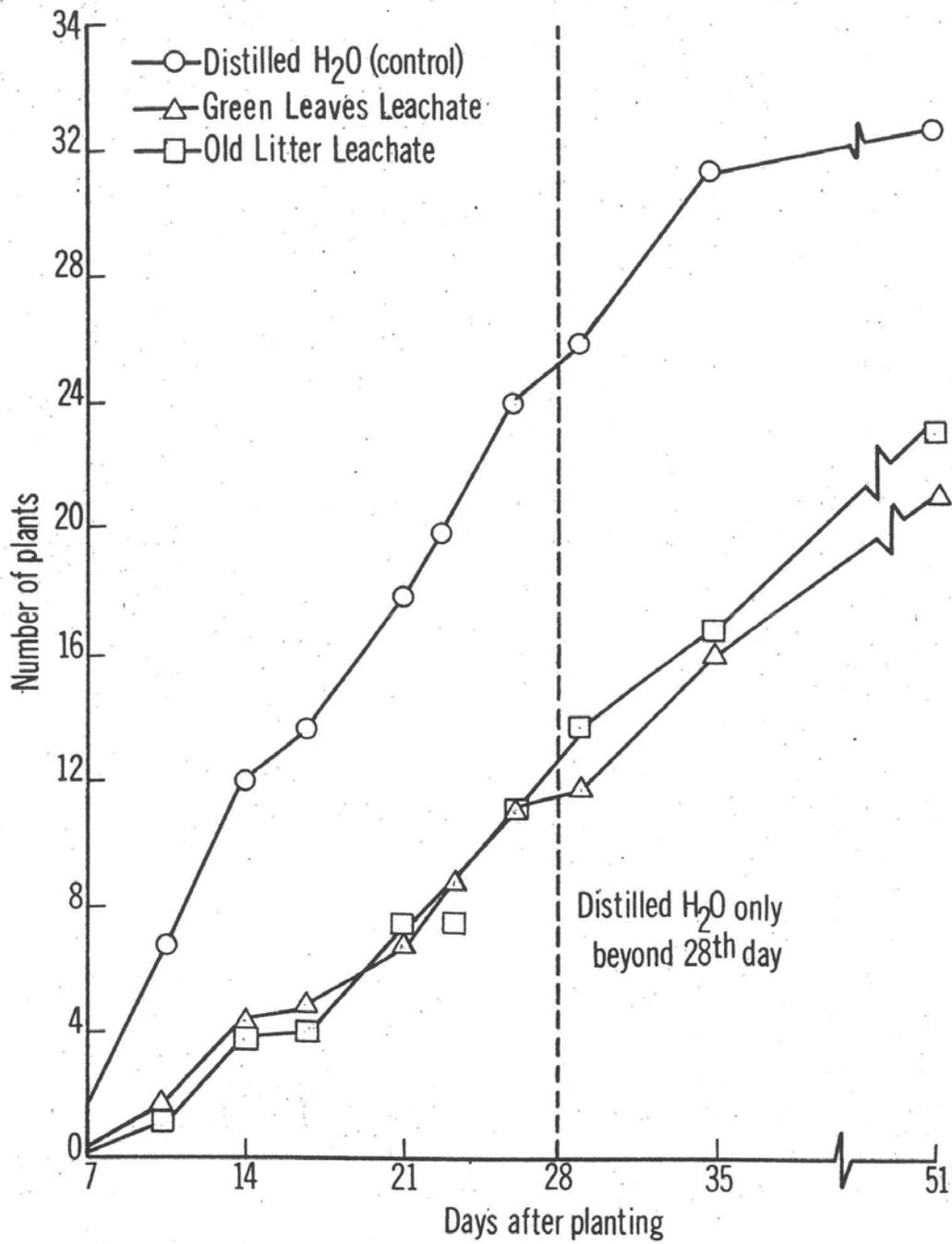


Figure 7 Emergence of Johnson grass plants from rhizomes under treatment with prairie grass leaf leachate (four replications per treatment).

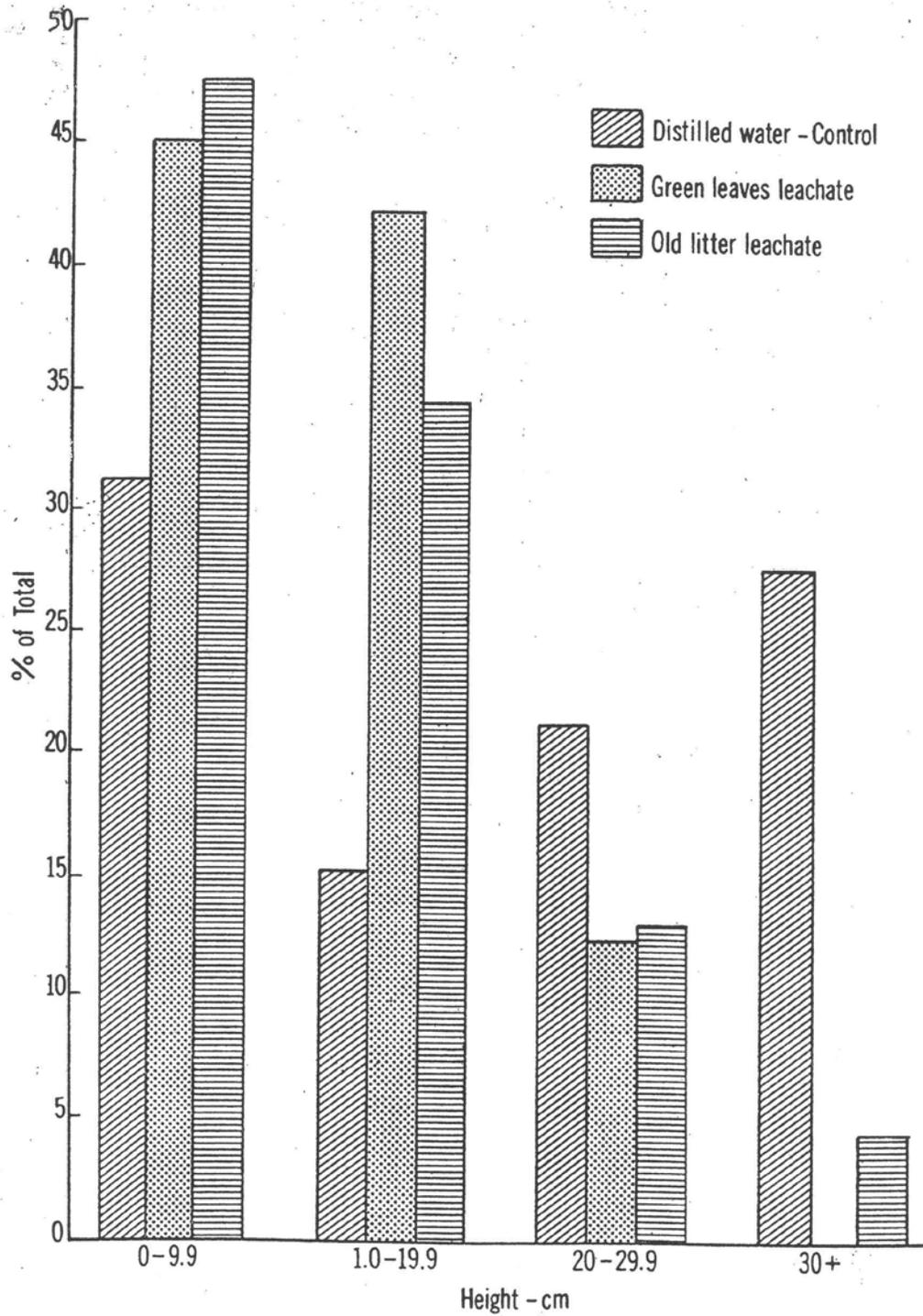


Figure 8 Height distribution of Johnson grass plants grown from rhizomes under treatments with prairie grass leaf leachate.

plants and litter. Johnson grass grew in a small, disturbed plot in a prairie but was stunted. Continued survival and establishment of the few Johnson grass plants that did grow were very doubtful. No Johnson grass growth was detected in the undisturbed or natural prairie plots.

Similar results were obtained with Johnson grass growth in Johnson grass stands. The only plants that emerged were in the disturbed or modified sites. The fact that none emerged in the plots in undisturbed Johnson grass stands might be expected. Abdul-Wahab and Rice (1967) reported that Johnson grass produced several inhibitory chemicals. Some of these inhibited its own seedling and rhizome bud growth. Upon observation, no young Johnson grass shoots were found within the stand. Numerous young plants were found along the edge of the stand spreading into the dirt road, but none were spreading out into the prairie side. The question remained of why no Johnson grass plants emerged in the undisturbed prairie.

Light intensity influenced Johnson grass growth. In the field, the only emergence was in the plots with either full sunlight or light shading. The most vigorous growth in the box experiment was obtained with light shading. With shading approximating that found at ground level in a prairie with heavy litter build-up, reduced growth of Johnson grass was noticed. Yet the dry weight of the Johnson grass plants after a whole season of growth in the disturbed prairie plots was considerably less than the dry weight of those under light shade after only 1 month of growth. The reduced emergence under the deep shading would not constitute exclusion. The leached litter produced average dry weights similar to those with deep shade but without the lower emergence. Aerial interference with prairie plants lowered both the average dry weight and emergence number of Johnson grass.

The few Johnson grass plants that grew when introduced in the small disturbed

prairie plots were small, weak, and stunted. In a box experiment, the Johnson grass plants growing near the prairie grasses were smaller and slightly discolored. Evidence suggests that the hypothesis that prairie grasses were producing some chemical inhibiting the growth of Johnson grass might be valid. The production of growth inhibiting substances by higher plants is not unknown. The production of these substances, termed allelopathic substances, appears to be widespread. Risser (1969) felt that allelopathic substances might play a part in formation and maintenance of vegetative patterns.

Some plants produce allelopathic substances that are known to be inhibitory to their own growth, as in the cases of *Bromus inermis*, *Helianthus pauciflorus*, *H. annuus*, and *Sorghum halepense* (Benedict 1941, Cooper and Stoesz 1931, Curtis and Cottam 1950, Wilson and Rice 1968, Abdul-Wahab and Rice 1967). Weaver and Rowland (1952) noted that the prairie grasses grew better with the removal of a heavy build-up of prairie mulch. They also remarked on the lack of understory herbs in a prairie with a heavy build-up of litter. An allelopathic substance in the grass litter would help explain the lack of understory vegetation. If the substance was short-lived once released or easily leached from shallow nursery flats, this would help explain the lag in emergence of Johnson grass in prairie soil or under prairie litter in previous experiments.

Since the inhibitory effect on Johnson grass was seen in the absence of root contact and in the presence of aerial parts, the leaves seemed a likely source. Something was present in the mixed leaves of little bluestem and Indian grass which inhibited bud growth of a Johnson grass rhizomal segment and the rate of plant growth. The inhibitory substance was present in both green leaves and dead litter. This indicated that sufficient quantity was present in the leaves to allow storage and slow release.

The implication existed that the inhibitory substance leached from the prairie grass might be influential in formation or maintaining of vegetative patterns in the prairie. Sagar and Harper (1961) showed that the presence and vigor of grasses in a community played a role in determining presence or absence of *Plantago* spp. Putwain and Harper (1970) concluded that the grasses were responsible for limiting population size of *Rumex* L. spp. The prairie grasses, little bluestem and Indian grass, seemed to play a role in restricting the growth of Johnson grass to along roadsides and out of the prairies.

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A PRELIMINARY PAWNEE ETHNOBOTANY CHECKLIST

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ABSTRACT

This document contains excerpts from a work in progress focusing on the ethnobotany of the Pawnee Native Americans. The effort being made is to consolidate research findings to provide a written record specifically addressing plant use by the Pawnee. The majority of the information gained was through literature reviews which provided a historic perspective. However, living among the Pawnee for twenty-two years has provided some insight into modern uses of some plants. A priority at the onset was to identify and describe the broad-ranging application of plants within their culture. All the ethnobotanical examples here are based on plants that have been documented in Oklahoma. Each plant is related to its currently known biogeography in Kansas and Nebraska which was regionally part of their historic homeland until their removal to Oklahoma beginning in 1875.

INTRODUCTION

Loss of land to encroaching Euro-Americans, inept government policies, disease, and warfare all contributed to the cultural degradation of the four bands or divisions of the Pawnee. The bands are known as the Chawi, Kitkahahki, Pitahawirata, and Skiri. The Pawnee belong to the Caddoan language family which also includes the Arikara, Caddo, and Wichita Tribes. Historically, the bands had linguistic differences and it is especially noted today when comparing the Skiri dialect to the often called, "south bands".

Well before the Tribe was relocated to what is now known as Pawnee, Oklahoma, cultural fragmentation had begun. For an account of the chronological history of the Pawnee see *The Pawnee Indians* by George E. Hyde (1951).

The floristic influence that enveloped and sustained the Pawnee culture in their homeland arose from prairie plant associations and riparian environments

linked to major rivers including the Loup, Platte, Republican, and their tributaries. In close proximity to their villages with earth lodges were gardens where they cultivated crops such as beans, corn, squash, and tobacco. Many plants were gathered from the surrounding areas to meet a variety of needs. In addition to farming, a summer and winter bison hunt was undertaken. Their survival and religious practices were critically dependent on a deep connection to the natural world.

Their new land allocation in Oklahoma consisted of different soils, a different climate and astronomical position, and plant life that further changed their life ways as pressure of acclimation and assimilation mounted. As a result, the botanical knowledge necessary to carry out ceremonies and other life ways continued to wane. As elders passed away, some loss of plant use knowledge accompanied them with each succeeding generation.

Although information sources dating in the 1800s were found that contributed to

the ethnobotany research, significant material was extracted from the works of James R. Murie (half Pawnee), Melvin R. Gilmore (married into the Tribe), George A. Dorsey, and others around the turn of the 20th century.

The intent of the research was not to limit the focus to plants used for food and medicine. Other relationships such as the use of plants for ceremonies, games, and materials were investigated to provide a broader representation as well as enhance an understanding of the complexity of Pawnee culture.

With each plant species listed, an attempt to document its location in Kansas, Nebraska, and Oklahoma was made primarily using the United States

Department of Agriculture (USDA) and Oklahoma Vascular Plants Database (OVPD) websites. The results revealed that certain plants were documented in all three states, other plants had limited or restricted ranges in one or more states, and some were not found in Oklahoma.

The presentation here consists of only a sample of plant species that are found in Oklahoma, reported to have been used by the Pawnee.

Providing a written record of consolidation specifically addressing the use of plants by the Pawnee may contribute to educational and cultural interests of the Pawnee Nation, individual Tribal members, and others.

PAWNEE ETHNIC BOTANY PLANT LISTING

Each plant is listed by family, genus, and species; former scientific name in parenthesis; one or more common names, including the Pawnee language name and meaning (if known); distribution of the plant; and information regarding usage of the plant.

CUPRESSACEAE

Juniperus virginiana L.

Eastern Redcedar or “Mother Cedar” (as referred to by the Pawnee)

Tawatsaako

Eastern redcedar grows in a variety of soil conditions and thrives in Kansas, Oklahoma, and Nebraska (USDA). True cedar, such as western redcedar, of the genus *Thuja*, is not native to Kansas, Oklahoma, and Nebraska.

This evergreen tree, commonly called “cedar”, was used by many Native Americans for a variety of purposes [Moerman 1998 (pp.290-291)]. In regard to the Pawnee, smoke inhaled from burned twigs is used as a remedy for colds; a decoction of fruits and leaves is used as a cough medicine and given to horses for same purpose; and boughs are put on tepee poles to ward off lightning (ibid).

Historically, juniper trees were used in ceremonies including the Skiri Doctors and Bear Dances [Murie 1981 (pp.170, 336)]. Skiri Bear Society participants used the leaves for ceremonial smudges, and if a thunderstorm should threaten, a smudge was made to protect the lodge (earth lodge) [Murie 1914 (p.604)]. Murie did not define “smudge” in the text cited. “Cedar poles” were fashioned for use as lances associated with the Two Lance Society [ibid (p.561)].

Gilmore [1919 (p.12)] noted that a smoke offering of cedar twigs was used as a remedy for nervousness and bad dreams. Based on my observations, I report that the Pawnee currently burn juniper leaves/needles as a ceremonial incense and/or prayer smoke offering, including use in the Native American Church. As I have witnessed, the smoke caused by placing juniper leaves on coals has been used to suffuse a person or object and in some situations only to allow smoke to permeate the surroundings. Prior to the practice, it is often said “going to burn some cedar” and the word “smudge” is not used. Nothing further will be added here since the focus of this paper is not to reveal ceremonial details.

According to Weltfish [1965 (p.387)], juniper was used for tepee poles by the Pawnee, but not necessarily as first choice if cottonwood was available.

Juniper also had a place in Ghost Dance hand game paraphernalia. Examples include hand game leaders Mark Rudder using a “cedar wood cross topped with a soft eagle feather and three cedar sticks topped with four red-painted crow feathers radially affixed to the top”, and Barclay White having used tally or counting sticks that were made of “cedar” in a ceremony [Lesser 1933 (pp.267-268, 288)].

Also, Emmett Pierson’s hand game set included a “sack of crumpled cedar leaves” that was part of the contents of a bundle. At the onset of the ceremony, a handful of the leaves were placed

on coals [Lesser 1933 (pp.274, 278)]. At one time, I was overseer of the Pierson Collection, on behalf of Skiri band member Ms. Maude Chisholm, which included a cloth bag containing juniper leaves. The collection on loan is housed in the Pawnee Bill Ranch Museum, Pawnee, Oklahoma, and also includes hand game sticks as well as other items.

AGAVACEAE

Yucca glauca Nutt.

Yucca or Soapweed Yucca
Chakida-kahtsu or Chakila-kahtsu

This perennial can be found growing in prairies and on hillsides in Kansas and Nebraska (USDA). It has been documented across Oklahoma (OVPD).

The yucca is known for the roots being used as soap, especially for washing hair, and the fibers from the leaves used by the Pawnee to make twine or cordage, with the leaf ends used as needles [Gilmore 1919 (p.19)]. I have made cordage from the leaves and found it to be quite strong, especially if primary use is for binding material.

LILIACEAE

Allium canadensis L. (=A. mutabile)

Wild Onion or Meadow Garlic
Osidawa

This perennial herb can be found growing in prairies, open woods, roadsides, and lawns. *Allium* can be found in Kansas and Nebraska (USDA). *A. canadensis* and other species of onion can be found throughout Oklahoma (OVPD). My personal experience of using “wild onions” as food is that a little can go a long way.

Moerman [1998 (p.57)] cited at least thirty species of *Allium* used by different Native American tribes, with reference to the above named species associated with the Pawnee for use as a spice, sauce, and relish. It is a species cited by Gilmore [1919 (p.19)], but under the older name, *A. mutabile*. Reportedly, the plant was eaten raw, cooked to flavor meat and soup, and also fried (ibid).

POACEAE

Arundinaria gigantea (Walter) Muhl.

River Cane or Giant Cane

USDA database lists the plant in Kansas, but not in Nebraska. More than 20 counties in eastern Oklahoma host the plant (OVPD).

River cane is the “bamboo” of North America. River cane served many purposes for tribes, especially the southeastern woodland cultures, in the making of arrows, blow guns, whistles, construction materials, fishing items, and in basketry [Moerman 1998 (p.104)]. The woody grass inhabits moist bottomlands and forest understories and can reach 20 or more feet in height.

Stewart Culin [1975 (p.99)], described Pawnee gaming sets containing four dice each that were made from split cane ranging from 8 to 16.5 inches (20.32 cm to 41.91 cm) in length, with some sets painted, and a set with a small feather tied to the end of each piece of cane.

According to Tuttle [1838 (p.41)] a type of flute was made out of cane which she noted as “sugar cane”. In my opinion, the flute was more likely made out of river cane. Moerman [1998 (p.499)] only listed the Seminole as using sugar cane as a food.

A small cane whistle was included in the warrior’s bundle belonging to Eagle Flying Under the Heavens [Murie 1981 (p.190)].

Hesperostipa spartea (Trin.) Barkworth (=Stipa spartea)
Porcupine or Needle Grass
Pitsuts (hair brush) or Paari pitsuts (Pawnee hairbrush)

Porcupine grass prefers dry prairies and open woods with a geographic range that includes Kansas and Nebraska. According to the USDA database and OVPD, the grass has been documented in three northern Oklahoma counties: Kay, Osage, and Washington. Pawnee County joins Osage County to the north.

The grass was prepared as a brush after the stiff awns and stalks were tightly bound into a small rounded-bundle, followed by burning off the pointed grains [Gilmore 1919 (p.14)]. Gilmore, citing Alice Fletcher, noted that the grass brush was used in the Pawnee Hako Pipe Ceremony.

Included in the publication by Fletcher [1904 (p.220)], regarding the Hako Ceremony, a section titled “Explanation by the Ku’rahus” (old man or priest) is as follows: “The grass of which the brush is made is gathered during a ceremony belonging to the Rain Shrine. It represents Toharu, the living covering of Mother Earth. The power which is in Toharu gives food to man and the animals so that they can live and become strong and able to perform the duties of life. This power represented by the brush of grass is now standing before the little child”. The grass brush was also described by Weltfish [1965 (p.363)].

TYPHACEAE

Typha latifolia L.

Cat-tail or Broadleaf Cattail

Hawahawa and Kirit-tacharush (meaning “eye itch” with reference to down getting into eyes)

The plant grows in a wide-spread range and conditions of moist sites and wetland environments. It can be found in Kansas, Oklahoma, and Nebraska (USDA and OVPD).

The Pawnee used the down to make dressings for burns and scalds [Moerman 1998 (p.574)]. Gilmore [1919 (p.12)] related that the down was used on infants to prevent chafing, as we use talcum; as a filling for pillows; and as padding for cradle boards, as well as in quilting baby wrappings. A great quantity of down was gathered in advance to have readied for the placement

of a newborn infant on the down. With the lack of cotton diapers in the olden times, pads of down were used (ibid).

Cat-tail leaves were used in the making of woven mats as an alternative to bulrush [Weltfish 1965(p.404)].

ANACARDIACEAE

Rhus glabra L.

Smooth Sumac

Nuppikt, sour top

Smooth sumac is a shrub that often grows in prairies, fields, and edges of woodlands. It is common across Oklahoma (OVPD) and much of Kansas and Nebraska (USDA).

The Pawnee name is in reference to the sour-tasting red fruits that develop in summer. In the fall when the leaves turn red, they were gathered and dried for smoking [Moerman 1998 (p.472)]. Gilmore [1919 (p.47)] also noted the use of the red leaves for smoking. In relation to a Chawi doctor ceremony, sumac leaves were mixed with tobacco for smoking [Murie 1981 (p.203)].

The fruits were boiled to make a remedy for dysmenorrhea and also for bloody flux [Gilmore 1919 (p.47)]. The use of its leaves, bark, and roots to make a black dye included the application to bison hides [Moerman 1998 (p.472)].

ARACEAE

Arisaema triphyllum (L.) Schott

Jack-In-The Pulpit or Indian Turnip

Nikso kororik kahtsu nitawau; medicine or herb, that bears,
what resembles, an ear of corn (the ripe fruits)

It grows in moist woodlands and is known to exist in Kansas and Nebraska (USDA). It has been documented in more than 17 counties in Oklahoma (OVPD).

If you eat the un-cooked corm (root), you will certainly reap the unpleasant sensation due to the calcium oxalate crystals inherent in this poisonous plant. The corm was not reportedly used as a food source of the Pawnee, but was pulverized and used as medicine. It was used to treat headaches by dusting the top of the head and temples and applied as a counterirritant for rheumatism and similar pains. The seeds were placed in gourd shell rattles [Gilmore 1919 (p.17)].

ASTERACEAE

Grindelia squarrosa (Push) Dun.

Curly cup or Curly top Gum weed

Bakskititis, stick-head (bak, head; skitits, sticky)

Gumweed is a perennial plant that grows in fields, along roadsides, and in waste places. The plant exudes a sticky substance which is true to the Pawnee name. It is spread across the

northern half of Kansas and found across the state of Nebraska (USDA). In Oklahoma, the plant is mostly situated in western counties (OVPD).

According to a Pawnee informant, the tops and leaves were boiled to make a wash for saddle galls and sores on horses [Gilmore 1919 (p.81)]. This species has the longest listing of uses by Native Americans of the *Grindelia* genus [Moerman 1998 (pp.252-253)].

***Helianthus annuus* L.**

Common Sunflower

Kirik-tara-kata, yellow eyes (kirik, eye; tara, having; kata, yellow)

The annual plant is often found in fields and along roadsides. The common sunflower is listed for every county in Kansas, and in Nebraska, it is distributed across the entire state (USDA). It has been documented in the majority of counties in Oklahoma (OVPD).

Gilmore [1919 (p.78)] noted that he could not find that the plant was ever cultivated by any of the Nebraska tribes, but there was evidence of such by some of the eastern tribes and the Arikara (linguistic neighbors to the north). A Pawnee informant of Gilmore's reported that the seeds were pounded up with certain roots (not identified or disclosed) and were taken in the dry form without further preparation, by women who became pregnant while still suckling a child for the reason that the suckling child should not become sick (ibid).

***Helianthus tuberosa* L.**

Jerusalem Artichoke

Kisu-sit (kisu, tapering; sit, long)

The perennial plant has been reported for mainly the eastern three-quarters of Kansas and Nebraska (USDA). In Oklahoma, it is erratically distributed mostly in the eastern half of the state (OVPD). It grows in wet soils of prairies, open woods, disturbed areas, and roadsides.

The people of the Nebraska tribes say they never cultivated the plant, but used its tubers for food [Gilmore 1919 (p.79)]. The Pawnee reportedly ate them only raw, but the others, according to their own statement, ate them raw, boiled, or roasted (ibid).

In a Pawnee tale, "Coyote and the Artichoke", artichoke (presumably Jerusalem artichoke), was mentioned; whereas, coyote ate too many artichokes which caused intestinal distress [Dorsey 1906 (p.464)]. Wonder what the moral of that story part is?

FABACEAE

***Apios americana* Medik. (=Glycine apios)**

Groundnut or Indian Potato

Its

The habitat of the perennial twining herb includes pond and stream banks, moist thickets, and wet meadows. It can be found in Kansas and Nebraska (USDA) and mostly in central, eastern, and some southwestern counties in Oklahoma (OVPD).

Two parts of the plant were a food source for the Pawnee. The tubers were eaten raw or cooked, preferably gathered in the fall, and the seeds of summer were consumed like peas [Kindscher 1987 (pp.48-49)]. Groundnut is a common native food plant of temperate and eastern North America. It is possible that the plant was propagated by the Cheyenne and other tribes and its range extended westward (ibid). Weltfish [1965 (p.415)] noted that the tubers were an important food provision for the winter bison hunt. Gilmore [1919 (p.42)] reported that the tubers of the plant were used as a food source by all the tribes within its range and prepared by boiling or roasting.

Pediomelum esculenta (Pursh) Rydb. (=Psoralea esculenta)

Indian Breadroot or Pomme Blanche

Patsuroka

The perennial herbaceous plant prefers prairies and has a scattered occurrence in about 16 Oklahoma counties (OVPD). It is scattered through much of Kansas and across Nebraska (USDA). The French name, “Pomme Blanche”, means white fruit.

The plant’s root was an important substance of the vegetal diet of the Plains tribes and after being peeled was eaten fresh, cooked, or stored to dry for use during the winter. The roots were braided in long strings by the tapering ends. When the women and children went to the prairie to gather the roots, on finding a plant the mother tells the children to note the directions which the several branches point and a child is sent in the general direction of each branch to look for another plant, for they say the plants “point to each other” [Gilmore 1919 (p.40)].

NELUMBONACEAE

Nelumbo lutea Willd.

Water Lily or Water Chinquapin

Tukawiu, Skiri band word and Tut, Chawi band

The aquatic plant currently has a range in Nebraska limited to 3 counties and is scattered in more than 20 counties in Kansas. It has been documented across Oklahoma (OVPD), but only in 12 counties by the USDA.

The plant was considered to be one invested with mystic powers. It was an important food source with use of the seeds and tubers (shaped somewhat like a banana). The hard, nutlike seeds were cracked and used with meat for making soup. The peeled tubers were cut up and cooked with meat or with hominy [Gilmore 1919 (p.27)].

RANUNCULACEA

Aquilegia canadensis L.

Wild Columbine or Red Columbine

Skalikatit or Skarikatit, black seed

The plant prefers growing in moist, well-drained, shady or partly shaded sites. It has been documented in some eastern Kansas counties and some northern and eastern counties in

Nebraska (USDA). In Oklahoma, the columbine is mainly in 17 northeastern and eastern counties (OVPD).

According to an account of the seeds being used as a perfume and a love charm, seeds are pulverized and rubbed in the palms of the suitor, who then contrives to shake hands with the desired one, whose fancy it is expected will thus be captivated [Gilmore 1919 (pp.30-31)]. Also, historically, seeds were crushed in an elm mortar by a pestle made of the same wood, with the resulting powder being added to hot water and the infusion being drunk for fever and headache (ibid).

CONCLUSION

I have made an attempt to provide a checklist of plant usage as it relates to the Pawnee. Before publication, a "Preliminary Pawnee Ethnobotany Checklist" was reviewed by Mr. Stephen Bird (B.S., M.S.) and three other Pawnees.

My goal is to complete the larger paper which may include more than sixty species of plants, not including species associated with agriculture. When that is done, a Pawnee review committee will be offered the opportunity to respond to the findings of the research.

Ethnobotany has many applications. Along with the existing Pawnee endeavors involving agriculture, linked to historic corn varieties and other cultivated plants of olden times, herbaceous native plants could also be grown in an ethnobotany garden to contribute to horticultural skills development, cultural education, and the actual use of the plants. Also, information gained through the research could be applied to the arts and in the sanctioned reproduction of certain artifacts. It is like filling ones tow sack with pieces of lost earthly connections to possess in order to bring elements of the past to the present.

Lastly, I share an excerpt from "Origin of the Chauí", also written "Chawí", as told by Roaming Chief-Hereditary Chief of the Chauí (band) in about 1906 and recorded by Dorsey [1997(p.13)]:

The earth I give you, and you are to call her 'mother', for she gives birth to all things. The timber that shall grow upon the earth you shall make use of in many ways. Some of the trees will have fruit upon them. Shrubs will grow from the ground and they will have berries upon them. All these things I give you and you shall eat of them. Never forget to call the earth 'mother', for you are to live upon her. You must love her, you must walk upon.

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VASCULAR FLORA OF ALABASTER CAVERNS STATE PARK, CIMARRON GYPSUM HILLS, WOODWARD COUNTY, OKLAHOMA

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ABSTRACT

Alabaster Caverns State Park is located in the Cimarron Gypsum Hills of northwestern Oklahoma, a semi-arid region of the state. The majority of the park is dominated by mixed-grass prairie and gypsum outcrops, with some riparian habitat and wooded north-facing slopes. A vascular plant inventory conducted from 2004 through 2007 yielded 274 species in 199 genera and 66 families. The largest families were the Poaceae (52 species), Asteraceae (47), and Fabaceae (23). There were 100 annuals, 6 biennials, and 163 perennials, as well as 5 species that have more than one life history form. Forty-two species (15.3%) were not native to North America. Three taxa currently being tracked by the Oklahoma Natural Heritage Inventory (2012) were present: *Echinocereus reichenbachii* (S3G5), *Haploesthes greggii* (S1G4?), and *Marsilea vestita* (S1G5). Compared to floristic inventories of sites in the Cimarron Gypsum Hills that are less impacted by public visitation, but more intensively grazed, Alabaster Caverns State Park has a higher number of species as well as a higher proportion of introduced species.

INTRODUCTION

Palmer et al. (1995) summarized the importance of floristic inventories in providing data for research on biodiversity, environmental impact assessment, and management decisions. The need for further studies of the vascular flora of the Gypsum Hills Physiographic Province was noted by Hoagland (2000). Since that time, two publications have provided floristic inventories of areas within the Cimarron Gypsum Hills of northwestern Oklahoma. Buckallew and Caddell (2003, 2004) summarized the vascular flora of the Selman Living Laboratory, located approximately 6 miles west of Alabaster Caverns State Park in Woodward County. It supports primarily mixed-grass prairie and gypsum outcrop

communities and was part of the Selman Ranch until 1998. Hoagland and Buthod (2005) surveyed a gypsum-dominated, currently-grazed ranch located approximately 24 miles southeast of Alabaster Caverns in Major County. Alabaster Caverns was established as a state park in the 1950s and therefore has a different land-use history. It is heavily visited by the public and is a site on the Western Oklahoma Wildlife Trail. The objectives of this inventory were to contribute to our knowledge of plant distributions in Oklahoma and in the Cimarron Gypsum Hills; to compare the vascular flora of Alabaster Caverns State Park to that of previously-described, more intensively-grazed but less heavily-visited sites in the Cimarron Gypsum Hills; and to provide a resource that

can be used by state park personnel for education and conservation purposes.

STUDY AREA

Alabaster Caverns State Park is located in Woodward County, Oklahoma (36°42'00"N, -99°08'47"W; T26N R18W SW1/4 of Sec. 28 and NW1/4 of NW1/4 of Sec. 33). The land for the park was purchased by the State of Oklahoma in 1953. It became a state park in 1956 (Allen 2007) and is managed by the Oklahoma Tourism and Recreation Department. The park consists of approximately 81 hectares (=200 acres). Cedar Creek, a tributary of Long Creek, flows west to east through Cedar Canyon and roughly bisects the park. Elevation ranges from about 488 m to 532 m.

The climate is semi-arid. According to climate data for Woodward County (Oklahoma Climatological Survey 2012), average annual precipitation is about 61 cm. The growing season lasts approximately 186 days, from mid-April to mid-October. The mean annual temperature is 15.6° C, with daily average temperatures ranging from 2.0° C in January to 27.8° C in July. Temperatures range from an average low of -5.6° C in January to an average daytime high of 35° C in July. Winds average 11 miles per hour and most often are from the south or southwest.

Alabaster Caverns lies in the Cimarron Gypsum Hills Province of Oklahoma (Curtis et al. 1979). Most of the park is underlain by the Blaine Formation, consisting of alternating layers of gypsum and shale formed during the Permian Period. The gypsum outcrops on the site belong to this formation. The Flowerpot Shale, which underlies the Blaine Formation, is exposed in Cedar Canyon (Meyers et al. 1969). Soils belong to the Vernon-Cottonwood Association and are excessively-drained loams and clay loams that have formed from gypsum and gypsiferous shales (Nance et al. 1963). The potential vegetation type is mixed grass (Duck and Fletcher 1943).

METHODS

We intensively surveyed the site throughout the growing seasons of 2004 and 2005. During those years, we visited the site 19 times, from May through October of 2004, and from April to October of 2005. We also surveyed the site in March and May of 2006. During most visits, we walked the areas both north and south of the canyon, and attempted to visit all major habitats within the park. We recorded all vascular plant species we encountered, noted whether they were in flower or fruit, and collected voucher specimens. We collected exotic species only from naturalized populations, excluding cultivated species from around the visitor center and campgrounds. A few species were identified by sight and documented only by photographs, generally because of their rarity at the site or their rarity status in Oklahoma. We added a few species to our vascular plant species list during plot sampling in 2006 and 2007 for a study of the vascular plant communities across the Cimarron Gypsum Hills (Rice 2008). References used for specimen identification included Hitchcock (1971), Great Plains Flora Association (1986), Diggs et al. (1999), Tyrl et al. (2005, 2010), and Barkworth et al. (2007). The organization of taxa in our species list is based on Angiosperm Phylogeny Group (APG III) recommendations (Stevens 2012), and nomenclature follows the PLANTS Database compiled by the United States Department of Agriculture, Natural Resources Conservation Service (USDA, NRCS 2012). The PLANTS Database was also used to determine whether each species was native to North America or introduced, and whether it was an annual, biennial, or perennial. In cases where species have more than one life form across their range, we noted the life form(s) encountered at Alabaster Caverns State Park. Voucher specimens were deposited in the University of Central Oklahoma (CSU) Herbarium.

RESULTS AND DISCUSSION

We identified 274 species in 199 genera and 66 families (Table 1, Appendix). These included 4 monilophytes (1 species of horsetail and 3 ferns), 1 gymnosperm, 210 eudicots, and 59 monocots. There was one additional subspecific taxon. Species in the Poaceae (52), Asteraceae (47), and Fabaceae (23) far outnumbered those in other families. Only 7 other families were represented by more than 5 species: Euphorbiaceae (11), Brassicaceae (8), Caryophyllaceae (7), Plantaginaceae (7), Solanaceae (7), Apocynaceae (6), and Onagraceae (6). The largest genera were *Astragalus* (6 species), *Oenothera* (6), *Chamaesyce* (5), and *Asclepias* (5). One hundred species were annuals, 6 were biennials, and 163 were perennials. Five species had more than one life form. Thirty-six species were trees (18 species), shrubs (12), or woody vines (6). *Cylindropuntia imbricata* is included on the species list because it apparently has escaped from cultivation within the park.

Three taxa tracked by the Oklahoma Natural Heritage Inventory (2012) were present: *Marsilea vestita* (S1G5), *Haploesthes greggii* (S1G4?), and *Echinocereus reichenbachii* (S3G5). Rarity ranks, in parentheses, range from 1 (critically imperiled) to 5 (demonstrably secure) at the state (S) and global (G) levels.

The park includes primarily mixed-grass prairie and gypsum outcrop plant communities. The major plant association (Hoagland 2000) is the *Schizachyrium scoparium*-*Castilleja purpurea* var. *citrina*-*Lesquerella gordonii* herbaceous association. The north-facing slopes are wooded, and the ravines of Cedar Canyon are dominated by *Juniperus virginiana*. The areas adjacent to the visitor center and within and adjacent to the park's two campgrounds are disturbed. Although the area south of the canyon has not been grazed since the 1950s, the area north of Cedar Canyon was leased for grazing until 1997 (Caywood 2006), and contains some old-field vegetation. Wetland and riparian vegetation is found

along Cedar Creek and on the edges of a pond near the western boundary of the park.

Forty-two species (15.3 %) in 16 families were not native to North America. Four of these species (*Bothriochloa ischaemum*, *Bromus tectorum*, *Sorghum halepense*, and *Tamarix ramosissima*) are listed as Oklahoma problem species, 4 (*Ailanthus altissima*, *Erodium cicutarium*, *Melilotus officinalis*, and *Ulmus pumila*) are on the Oklahoma Watch List, and 14 are problems in border states (Oklahoma Invasive Plants Council 2012). Seventeen species of Poaceae were introduced.

Compared with the recently-grazed Selman Living Lab (Buckallew and Caddell 2003, 2004) and the currently-grazed Major County ranch (Hoagland and Buthod 2005), Alabaster Caverns State Park had a higher number of plant species, although it is smaller (81 ha) than the Selman Living Lab (129.5 ha) and approximately the same size as the Major County ranch (80+ ha). The higher number of species is in part due to the higher number and proportion of introduced species at Alabaster Caverns. Of the 229 species at the Selman Living Lab, 21 (9%) were introduced. Of the 233 species at the Major County ranch, 22 (10.6%) were introduced. The higher number of introduced species at Alabaster Caverns can be attributed to disturbance associated with the high number of visitors to the park, especially around the visitor center and campgrounds. Of the 274 species at Alabaster Caverns State Park, 175 also occur at the Selman Living Lab. Of the 99 species that occur at Alabaster Caverns but not at the Selman Living Lab, 33 are introduced species. Other differences in species composition are due to differences in land-use history and habitats between the two sites; the Selman Living Lab had been recently grazed when it was inventoried, and it includes sandsage grassland as well as a much larger floodplain than Alabaster Caverns State Park. Although the northern part of Alabaster Caverns State Park was grazed recently, the southern part has not been grazed since the 1950s. Because the Selman Living Lab is located only 6 miles

to the west of Alabaster Caverns State Park, annual temperature and precipitation are the same, and therefore do not contribute to differences in species composition. Alabaster Caverns State Park shares 163 species with the Major County ranch. Differences in species composition between those 2 sites can be attributed in part to their different grazing histories as well as to some differences in habitats. The Major County Ranch is grazed currently, and contains a large pond, disturbed areas associated with oil well pads, and more roads than Alabaster Caverns State Park. Environmental factors also differ between the sites. Although average temperature differs by only 1° C, average annual precipitation is approximately 61 cm for Alabaster Caverns State Park and approximately 70 cm for the Major County ranch.

The major vegetation associations at Alabaster Caverns and brief descriptions of common species are as follows:

1. *Schizachyrium scoparium*-*Castilleja purpurea* var. *citrina*-*Lesquerella gordonii* herbaceous association

This was the predominant vegetation association in the park, on the gypsum outcrops and shallow soils on gypsum (Figures 1-3). Common associated species included *Aristida purpurea*, *Bouteloua curtipendula*, *Bouteloua gracilis*, *Chamaesyce glyptosperma*, *Croton monanthogynous*, *Dalea enneandra*, *Echinocereus reichenbachii*, *Heterotheca stenophylla*, *Lithospermum incisum*, *Mentzelia nuda*, *Mentzelia oligosperma*, *Nama stevensii*, *Oenothera hartwegii*, *Oenothera serrulata*, *Opuntia phaeacantha*, *Paronychia jamesii*, *Phacelia integrifolia*, *Polanisia dodecandra*, *Polygala alba*, *Portulaca pilosa*, *Psilostrophe tagetina*, *Sporobolus cryptandrus*, *Thelesperma magapotamicum*, *Tridens muticus* var. *elongatus*, and *Yucca glauca*. Two of these species, *Phacelia integrifolia* (Figure 4) and *Nama stevensii* (Figure 5), as well as the less-commonly encountered *Haploesthes greggii* (Figure 6), are found only on gypsum substrates in Oklahoma and are considered obligate gypsophiles. Two of the

species in this habitat, *Echinocereus reichenbachii* (Figure 7) and *Haploesthes greggii*, are being tracked by the Oklahoma Natural Heritage Inventory. Woody species occurred mainly on the steep north-facing slopes and ravines of Cedar Canyon, and included *Celtis laevigata* var. *reticulata*, *Cornus drummondii*, *Gleditsia triacanthos*, *Juniperus virginiana*, *Morus rubra*, *Rhus glabra*, *Rhus aromatica*, *Ribes aureum*, *Sapindus saponaria*, *Sideroxylon lanuginosum*, *Symphoricarpos orbiculatus*, *Ulmus americana*, *Ulmus rubra*, and *Vitis acerifolia*.

2. Wetland and riparian vegetation

This vegetation was found along the banks of Cedar Creek as well as the margins of the pond. Associated species included *Amorpha fruticosa*, *Baccharis salicina*, *Carex grvida*, *Eleocharis montevidensis*, *Equisetum* spp., *Nasturtium officinale*, *Pluchea odorata*, *Populus deltoides*, *Ranunculus sceleratus*, *Salix nigra*, and *Vitis riparia*. A wet depression in the grassland on the north side of the canyon supported *Marsilea vestita*, a species tracked by the Oklahoma Natural Heritage Inventory.

3. Disturbed areas and old-field vegetation

This type of vegetation was found in disturbed areas along roadsides and trails near the visitor center, in campgrounds, and in areas with deeper soils north of the canyon that were grazed until 1997. Common species in disturbed areas along roadsides, trails, and campgrounds were *Arenaria serpyllifolia*, *Bothriochloa ischaemum*, *Bromus* spp., *Chamaesaracha coniodes*, *Digitaria ciliaris*, *Erodium cicutarium*, *Glandularia pumila*, *Holosteum umbellatum*, *Lamium amplexicaule*, *Melilotus officinalis*, *Quincula lobata*, *Veronica* spp., and *Tribulus terrestris*. Common species in old fields included *Ambrosia psilostachya*, *Amphiachyris dracunculoides*, *Aristida oligantha*, *Artemisia ludoviciana*, *Bothriochloa laguroides*, *Bromus* spp., *Chamaesyce* spp., and *Gutierrezia sarothrae*. Many of these species increase with grazing. Thickets of *Prunus angustifolia* were also present.

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Table 1 Summary of floristic collections from Alabaster Caverns State Park in the Cimarron Gypsum Hills, Woodward County, Oklahoma*

Taxonomic Group	Families	Genera	Species	Native spp.	Exotic spp.
Monilophyta	3	4	4	4	0
Pinophyta	1	1	1	1	0
Magnoliophyta					
Eudicots	57	151	210	185	25
Monocots	5	43	59	42	17
Total	66	199	274	232	42

*Table format follows Palmer et al. (1995)

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APPENDIX

Annotated species list for Alabaster Caverns State Park, Woodward County, Oklahoma. Nomenclature and common names are based on USDA, NRCS (2012). Organization of taxa is based on Angiosperm Phylogeny Group (APG III) recommendations (Stevens 2012). Life history (A=annual, B=biennial, P=perennial) and collection numbers follow the species names. Taxa introduced to North America are indicated with an asterisk (*) and those on the Oklahoma Natural Heritage Inventory Plant Tracking List are indicated with a symbol (+). Voucher specimens were deposited in the University of Central Oklahoma Herbarium (CSU).

MONILOPHYTA

Equisetaceae

Equisetum L. sp. (horsetail) – P; GMC1215

Marsileaceae

+*Marsilea vestita* Hook & Grev. (hairy waterclove) – P; GMC1145

Pteridaceae

Cheilanthes feei T. Moore (slender lipfern) – P; GMC800

Pellaea atropurpurea (L.) Link (purple cliffbreak) – P; GMC815

GYMNOSPERMS/PINOPHYTA

Cupressaceae

Juniperus virginiana L. var. *virginiana* (eastern redcedar) – P; GMC816

ANGIOSPERMS/MAGNOLIOPHYTA

EUDICOTS

Amaranthaceae

Amaranthus tuberculatus (Moq.) Sauer (roughfruit amaranth) – A; GMC1245

**Chenopodium album* L. var. *album* (lambsquarters) – A; KR930

Chenopodium berlandieri Moq. (pitseed goosefoot) – A; GMC1217

Anacardiaceae

Rhus aromatica Aiton – P; GMC811

Rhus copallinum L. (winged sumac) – P; GMC1177

Rhus glabra L. (smooth sumac) – P; GMC849

Toxicodendron radicans (L.) Kuntze (eastern poison ivy) – P; GMC1267

Apiaceae

Ammoselinum popei Torr. & A. Gray (plains sandparsley) – A; KR753

Sanicula canadensis L. (Canadian blacksnakeroot) – B; GMC1170

Spermolepis inermis (Nutt. ex DC.) Mathias & Constance (Red River scaleseed) – A; GMC1165

Apocynaceae

Apocynum cannabinum L. (Indianhemp) – P; GMC 1137

Asclepias asperula (Decne.) Woodson ssp. *capricornu* (Woodson) Woodson
(antelopehorns) – P; GMC1096

Asclepias engelmanniana Woodson (Engelmann's milkweed) – P; GMC1186
Asclepias latifolia (Torr.) Raf. (broadleaf milkweed) – P; GMC1189
Asclepias viridiflora Raf. (green comet milkweed) – P; GMC870
Asclepias viridis Walter (green antelopehorn) – P; GMC1136

Asteraceae

Achillea millefolium L. (common yarrow) – P; GMC1107
Ambrosia psilostachya DC. (Cuman ragweed) – P; GMC897
Ambrosia trifida L. (great ragweed) – A; GMC914
Amphiachyris dracunculoides (DC.) Nutt. (prairie broomweed) – A; GMC922
Artemisia dracunculus L. (tarragon) – P; GMC921
Artemisia filifolia Torr. (sand sagebrush) – P; GMC895
Artemisia ludoviciana Nutt. ssp. *ludoviciana* (white sagebrush) – P; GMC941
Baccharis salicina Torrey & A. Gray (willow baccharis) – P; GMC901
Brickellia eupatorioides (L.) Shinnery var. *corymbulosa* (Torr. & A. Gray) Shinnery
(false boneset) – P; GMC1242
Chaetopappa ericoides (Torr.) G. L. Nesom (rose heath) – P; GMC1063
Cirsium undulatum (Nutt.) Spreng. (wavy leaf thistle) – P; GMC1161
Conyza canadensis (L.) Cronquist (Canadian horseweed) – A; KR929
Conyza ramosissima Cronquist (dwarf horseweed) – A; GMC1256
Echinacea angustifolia DC. (blacksamson echinacea) – P; GMC1136
Erigeron cf. *divergens* Torr. & A. Gray (spreading fleabane) – B; GMC973
Erigeron strigosus Muhl. ex Willd. (prairie fleabane) – A; GMC1097
Evax prolifera Nutt. ex DC. (bighead pygmyweed) – A; GMC1030
Gaillardia pulchella Foug. (Indian blanket) – A; GMC828
Gaillardia suavis (A. Gray & Engelm.) Britton & Rusby (perfumeballs) – P; GMC1133
Grindelia papposa G. L. Nesom and Suh (Spanish gold) – A; GMC1203
Grindelia squarrosa (Pursh) Dunal (curlycup gumweed) – B; GMC935
Gutierrezia sarothrae (Pursh) Britton & Rusby (broom snakeroot) – P; GMC907
+ *Haploesthes greggii* A. Gray (false broomweed) – P; GMC1147
Helianthus annuus L. (common sunflower) – A; GMC854
Helianthus petiolaris Nutt. (prairie sunflower) – A; GMC1244
Heterotheca stenophylla (A. Gray) Shinnery (stiffleaf false goldenaster) – P; GMC891
Hymenopappus tenuifolius Pursh (Chalk Hill hymenopappus) – B; GMC1128
Iva annua L. (annual marshelder) – A; GMC1257
Lactuca ludoviciana (Nutt.) Riddell (biannual lettuce) – B; GMC814
Liatris punctata Hook. (dotted blazing star) – P; GMC926
Machaeranthera pinnatifida (Hook.) Shinnery (tansyaster) – P; GMC1160
Packera plattensis (Nutt.) W.A. Weber & Á. Löve (prairie groundsel) – B, P; GMC1104
Pluchea odorata (L.) Cass. (sweetscent) – A; GMC1251
Psilostrophe tagetina (Nutt.) Greene var. *cerifera* (A. Nelson) B. L. Turner (woolly
paperflower) – P; GMC843
Pyrrhopappus grandiflorus (Nutt.) Nutt. (tuberous desert-chicory) – P; GMC1005
Ratibida columnifera (Nutt.) Woot. & Standl. (upright prairie coneflower) – P; GMC1113
Senecio riddellii Torr. & A. Gray (Riddell's ragweed) – P
Solidago missouriensis Nutt. var. *fasciculata* Holz (Missouri goldenrod) – P; GMC1220
Solidago petiolaris Aiton (downy ragged goldenrod) – P; GMC908

- **Sonchus asper* (L.) Hill (spiny sowthistle) – A; GMC1142
Symphyotrichum ericoides (L.) G. L. Nesom (white heath aster) – P; GMC944
 **Taraxacum officinale* F. H. Wigg (common dandelion) – P; GMC822
Tetranneuris scaposa (DC.) Greene (stemmy four-nerve daisy) – P; GMC1053
Thelesperma megapotamicum (Spreng.) Kuntze (Hopi tea greenthread) – P; GMC803
 **Tragopogon dubius* Scop. (yellow salsify) – B; GMC1143
Vernonia baldwinii Torr. (Baldwin's ironweed) – P; GMC900, GMC864
Xanthium strumarium L. var. *canadense* (Mill.) Torr. & Gray (Canada cocklebur) – A; GMC1253

Boraginaceae

- Lappula occidentalis* (S. Watson) Greene (flatspine stickseed) – A; KR752
Lithospermum incisum Lehm. (narrowleaf stoneseed) – P; GMC968

Brassicaceae

- **Camelina rumelica* Velen. (graceful false flax) – A; GMC1089
 **Capsella bursa-pastoris* (L.) Medik. (shepherd's purse) – A; GMC979
Descurainia pinnata (Walter) Britton (western tansymustard) – A; GMC1031
Draba reptans (Lam.) Fernald (Carolina draba) – A; GMC965
Lepidium densiflorum Schrad. (common pepperweed) – A; GMC1086
Lepidium oblongum Small (veiny pepperweed) – A, B; GMC1081, GMC963
Lesquerella gordonii (A. Gray) S. Watson (Gordon's bladderpod) – A; GMC966, GMC1057
 **Nasturtium officinale* W.T. Aiton (watercress) – P; GMC1179

Cactaceae

- Cylindropuntia imbricata* (Haw.) F.M.Knuth (tree cholla) – P
Echinocereus reichenbachii (Terscheck ex Walp.) hort ex Haage (lace hedgehog cactus) – P
Escobaria missouriensis (Sweet) D.R. Hunt (Missouri foxtail cactus) – P; GMC1195
Escobaria vivipara (Nutt.) Buxbaum var. *vivipara* (spinystar) – P; GMC1164
Opuntia phaeacantha Engelm. (tulip pricklypear) – P; GMC1144

Campanulaceae

- Triodanis perfoliata* (L.) Nieuwl. (clasping Venus' looking-glass) – A; GMC1122

Cannabaceae

- Celtis laevigata* Willd. var. *laevigata* (sugarberry) – P; GMC1071
Celtis laevigata Willd. var. *reticulata* (Torr.) L.D. Benson (netleaf hackberry) – P; GMC917
Celtis occidentalis L. (common hackberry) – P; GMC804

Caprifoliaceae

- Symphoricarpos orbiculatus* Moench (coralberry) – P; GMC915

Caryophyllaceae

- **Arenaria serpyllifolia* L. (thymeleaf sandwort) – A; GMC821
Cerastium nutans Raf. (nodding chickweed) – A; GMC1064
 **Cerastium pumilum* W. Curtis (European chickweed) – A; GMC1039
 **Holosteum umbellatum* L. (jagged chickweed) – A; GMC976
Paronychia jamesii Torr. & A. Gray (James' nailwort) – P; GMC883

Silene antirrhina L. (sleepy silene) – A; GMC1123

**Stellaria media* (L.) Vill. ssp. *pallida* (Dumort) Asch. & Graebon. (common chickweed) – A; GMC1014

Celastraceae

Celastrus scandens L. (American bittersweet) – P; GMC1002

Cleomaceae

Polanisia dodecandra (L.) DC. (redwhisker clammyweed) – A; GMC868

Clusiaceae

**Hypericum perforatum* L. (common St. Johnswort) – P; GMC1166

Convolvulaceae

Evolvulus nuttallianus Schult. (shaggy dwarf morning-glory) – P; GMC1088

Ipomoea leptophylla Torr (bush morning-glory) – P; GMC1146

Cornaceae

Cornus drummondii C.A. Mey. (roughleaf dogwood) – P; GMC799

Cucurbitaceae

Cucurbita foetidissima Kunth (Missouri gourd) – P; GMC1172

Euphorbiaceae

Acalypha ostryifolia Riddell (pineland threeseed mercury) – A; GMC927

Chamaesyce stictospora (Engelm.) Small (slimseed sandmat) – A; GMC892

Chamaesyce glyptosperma (Engelm.) Small (ribseed sandmat) – A; GMC1219

Chamaesyce maculata (L.) Small (spotted sandmat) – A; GMC1241

Chamaesyce missurica (Raf.) Shinnery (prairie sandmat) – A; GMC869

Chamaesyce serpens (Kunth) Small (matted sandmat) – A; GMC1259

Croton monanthogynus Michx. (prairie tea) – A; GMC930

Croton texensis (Klotzsch) Mull. Arg. (Texas croton) – A; GMC886, GMC862, GMC902

Euphorbia dentata Michx. (toothed spurge) – A; GMC953

Euphorbia marginata Pursh (snow on the mountain) – A; GMC937

Euphorbia spathulata Lam. (warty spurge) – A; GMC1060

Fabaceae

Amorpha canescens Pursh (leadplant) – P; GMC825

Amorpha fruticosa L. (false indigo bush) – P; GMC840

Astragalus gracilis Nutt. (slender milkvetch) – P; GMC993

Astragalus lotiflorus Hook. (lotus milkvetch) – P; GMC967, GMC992

Astragalus missouriensis Nutt. (Missouri milkvetch) – P; GMC1092, GMC1269, GMC969

Astragalus mollissimus Torr. (woolly locoweed) – P; GMC1093

Astragalus nuttallianus DC. var. *austrinus* (Small) Barneby (smallflowered milkvetch) – A; GMC1049

Astragalus plattensis Nutt. (Platte River milkvetch) – P; GMC1046, GMC1047, GMC1099

Dalea aurea Nutt. ex Pursh (golden prairie clover) – P; GMC863

Dalea candida Michx. ex Willd. var. *candida* (white prairie clover) – P; GMC866

Dalea enneandra Nutt. (nineanther prairie clover) – P; GMC1154

Dalea purpurea Vent. (purple prairie clover) – P; GMC1153
Desmanthus illinoensis (Michx.) MacMill. ex B.L. Rob. & Fernald (Illinois bundleflower) – P; GMC924
Gleditsia triacanthos L (honeylocust) – P; GMC986
 **Medicago minima* (L.) L. (little bur-clover) – A; GMC1043
 **Melilotus officinalis* (L.) Lam. (sweetclover) – A,B; GMC827, GMC850
Mimosa quadrivalvis L. (fourvalve mimosa) – P; GMC1090
Pediomelum cuspidatum (Pursh) Rydb. (largebract Indian breadroot) – P; GMC1091, GMC 1135
Prosopis glandulosa Torr. (honey mesquite) – P; GMC932
Psoraleidium tenuiflorum (Pursh) Rydb. (slimflower scurfpea) – P; GMC1169
Robinia pseudoacacia L. (black locust) – P; GMC1070
Vicia americana Muhl. ex Willd. (American vetch) – P; GMC1000
Vicia ludoviciana Nutt. (Louisiana vetch) – A; GMC1094

Fagaceae

Quercus muehlenbergii Engelm. (chinkapin oak) – P

Geraniaceae

**Erodium cicutarium* (L.) L'Her. ex Aiton (redstem stork's bill) – A; GMC 836
 **Geranium pusillum* L. (small geranium) – A; 1020

Grossulariaceae

Ribes aureum Pursh var. *villosum* DC. (golden currant) – P; GMC971

Hydrophyllaceae

Nama stevensii C.L. Hitchc. (Steven's fiddleleaf) – A; GMC1041
Phacelia integrifolia Torr. (gyp phacelia) – A,B; GMC1187

Lamiaceae

Hedeoma hispida Pursh (rough false pennyroyal) – A; GMC795
 **Lamium amplexicaule* L. (henbit deadnettle) – A; GMC981
Monarda clinopodioides A. Gray (basil beebalm) – A; GMC1159
Teucrium laciniatum Torr. (lacy germander) – P; GMC1134

Linaceae

Linum pratense (Norton) Small (meadow flax) – A; GMC1066
Linum rigidum Pursh (stiffstem flax) – A; GMC1067

Loasaceae

Mentzelia nuda (Pursh) Torr. & A. Gray var. *stricta* (Osterh.) Harrington (bractless blazingstar) – B,P; GMC1188
Mentzelia oligosperma Nutt. ex Sims (chickenthiel) – P; GMC802

Malvaceae

Callirhoe involucrata (Torr. & A. Gray) A. Gray (purple poppymallow) – P; GMC1006
Sphaeralcea coccinea (Nutt.) Rydb. (scarlet globemallow) – P; GMC1051

Molluginaceae

Mollugo verticillata L. (green carpetweed) – A; GMC1229

Moraceae

Morus rubra L. (red mulberry) – P; GMC1138

Nyctaginaceae

Mirabilis linearis (Pursh) Heimerl (narrowleaf four o'clock) – P; GMC1180

Mirabilis nyctaginea (Michx.) MacMill. (heartleaf four o'clock) – P; GMC1139

Oleaceae

Forestiera pubescens Nutt. (stretchberry) – P; GMC1249

Onagraceae

Oenothera cinerea (Wooton & Standl.) W.L. Wagner & Hoch (woolly beeblossom) – P; GMC809

Oenothera curtiflora W.L. Wagner & Hoch (velvetweed) – A; GMC1148

Oenothera glaucifolia W.L. Wagner & Hoch (false gaura) – P; GMC807

Oenothera hartwegii Benth. (Hartweg's sundrops) – P; GMC1127

Oenothera serrulata Nuttall (yellow sundrops) – P; GMC1110

Oenothera suffrutescens (Ser.) W.L. Wagner & Hoch (scarlet beeblossom) – P; GMC1052, GMC1126

Orobanchaceae

Agalinis aspera (Douglas ex Benth.) Britton (tall false foxglove) – A; GMC1228

Castilleja purpurea (Nutt.) G. Don var. *citrina* (Pennell) Shinnery (prairie Indian paintbrush) – P;
GMC991

Orobanche ludoviciana Nutt. ssp. *multiflora* (Nutt.) T.S. Collins ex H.L. White & W.C.
Holmes (manyflower broomrape) – A; GMC1196

Oxalidaceae

Oxalis corniculata L. (creeping woodsorrel) – A; GMC983

Oxalis dillenii Jacq. (slender yellow woodsorrel) – P; GMC1019

Papaveraceae

Argemone polyanthemos (Fedde) G.B. Ownbey (crested pricklypoppy) – A

Corydalis micrantha (Engelm. ex A. Gray) A. Gray (smallflower fumewort) – A; GMC1271

Plantaginaceae

Nuttallanthus canadensis (L.) D.A. Sutton (Canada toadflax) – A; KR441

Penstemon cobaea Nutt. (cobaea beardtongue) – P; GMC1056

Plantago patagonica Jacq. (woolly plantain) – A; GMC1087

Plantago rhodosperma Decne. (redseed plantain) – A; GMC1062

**Veronica arvensis* L. (corn speedwell) – A; GMC1045

Veronica peregrina L. ssp. *xalapensis* (Kunth) Pennell (hairy purslane speedwell) – A; GMC964

**Veronica polita* Fr. (gray field speedwell) – A; GMC 984

Polygalaceae

Polygala alba Nutt. (white milkwort) – P; GMC865

Polygonaceae

**Polygonum persicaria* L. (spotted ladythumb) – A; GMC1190

Polygonum ramosissimum Michx. (bushy knotweed) – A; GMC1191

Rumex altissimus Alph. Wood (pale dock) – P; GMC1209

Portulacaceae

**Portulaca oleracea* L. (little hogweed) – A; GMC1232

Portulaca pilosa L. (kiss me quick) – A; GMC925

Primulaceae

Androsace occidentalis Pursh (western rockjasmine) – A; GMC1272

Ranunculaceae

Delphinium carolinianum Walter ssp. *virescens* (Nutt.) R.E. Brooks (Carolina larkspur) – P

Ranunculus sceleratus L. (cursed buttercup) – A

Rhamnaceae

Ceanothus herbaceus Raf. (Jersey tea) – P; GMC1106

Rosaceae

Prunus angustifolia Marsh. (Chickasaw plum) – P; GMC844, GMC972

Rubiaceae

Galium aparine L. (stickywilly) – A; GMC1003

Galium circaezans Michx. (licorice bedstraw) – P; GMC1193

Stenaria nigricans (Lam.) Terrell var. *nigricans* (prairie bluet) – P; GMC1167

Salicaceae

Populus deltoides Bartram ex Marsh. (eastern cottonwood) – P

Salix nigra Marsh. (black willow) – P; GMC997

Sapindaceae

Sapindus saponaria L. var. *drummondii* (Hook. and Arn.) L.D. Benson (western soapberry) – P;
GMC1206

Sapotaceae

Sideroxylon lanuginosum Michx. (gum bully) – P; GMC835, GMC1207

Simaroubaceae

**Ailanthus altissima* (Mill.) Swingle (tree of heaven) – P

Solanaceae

Chamaesaracha coniodes (Moric. ex Dunal) Britton (gray five eyes) – P; GMC1044

Physalis cf. *hederifolia* A. Gray (ivyleaf groundcherry) – P; GMC857

Physalis longifolia Nutt. (longleaf groundcherry) – P; GMC1205

Physalis mollis Nutt. (field groundcherry) – P; GMC1216

Quincula lobata (Torr.) Raf. (Chinese lantern) – P; GMC1085

Solanum elaeagnifolium Cav. (silverleaf nightshade) – P; GMC896
Solanum rostratum Dunal (buffalobur nightshade) – A; GMC936

Tamaricaceae

**Tamarix ramosissima* Ledeb. (saltcedar) – P; GMC1192

Ulmaceae

Ulmus americana L. (American elm) – P; GMC970
**Ulmus pumila* L. (Siberian elm) – P; GMC978
Ulmus rubra Muhl. (slippery elm) – P

Urticaceae

Parietaria pensylvanica Muhl. ex Willd. (Pennsylvania pellitory) – A

Verbenaceae

Glandularia bipinnatifida (Nutt.) Nutt. (Dakota mock vervain) – P; GMC1050
Glandularia pumila (Rydb.) Umber (pink mock vervain) – A; GMC830

Violaceae

Viola bicolor Pursh (field pansy) – A; GMC962

Vitaceae

Cissus trifoliata (L.) L. (sorrelvine) – P; GMC845
Parthenocissus quinquefolia (L.) Planch. (Virginia creeper) – P; GMC823
Vitis acerifolia Raf. (mapleleaf grape) – P; GMC1175
Vitis riparia Michx. (riverbank grape) – P; GMC826, GMC1208

Zygophyllaceae

**Tribulus terrestris* L. (puncturevine) – A; GMC1198

MONOCOTS

Amaryllidaceae

Allium drummondii Regel (Drummond's onion) – P; GMC987

Asparagaceae

Androstephium coeruleum (Scheele) Greene (blue funnel lily) – P; GMC974
Yucca glauca Nutt. var. *glauca* (soapweed yucca) – P; GMC1061

Commelinaceae

Tradescantia occidentalis (Britton) Smyth (prairie spiderwort) – P; GMC1095

Cyperaceae

Carex gravida L.H. Bailey (heavy sedge) – P; GMC838
Cyperus lupulinus (Spreng.) Marcks (Great Plains flatsedge) – P; GMC929
Eleocharis montevidensis Kunth (sand spikerush) – P; GMC1273

Poaceae

- **Aegilops cylindrica* Host (jointed goatgrass) – A; GMC1108
Andropogon hallii Hack. (sand bluestem) – P; GMC950
Aristida oligantha Michx. (prairie threeawn) – A; GMC1221, GMC1262
Aristida purpurea Nutt. (purple threeawn) – P; GMC861
 **Bothriochloa ischaemum* (L.) Keng (yellow bluestem) – P; GMC955
Bothriochloa laguroides (DC.) Herter ssp. *torreyana* (Steud.) Allred & Gould (silver beardgrass) – P; GMC1162
Bouteloua curtipendula (Michx.) Torr. (sideoats grama) – P; GMC846
Bouteloua gracilis (Willd. ex Kunth) Lag. ex Griffiths (blue grama) – P; GMC872
Bouteloua hirsuta Lag. (hairy grama) – P; GMC884
 **Bromus catharticus* Vahl (rescuegrass) – A; GMC989
 **Bromus arvensis* L. (field brome) – A; GMC1119
 **Bromus tectorum* L. (cheatgrass) – A; GMC1124, GMC988
Buchloe dactyloides (Nutt.) J.T. Columbus (buffalograss) – P; GMC1116, GMC1027, GMC960
Cenchrus spinifex Cav. (coastal sandbur) – A; GMC834
Chloris verticillata Nutt. (tumble windmill grass) – P; GMC1231
 **Dactylis glomerata* L. (orchardgrass) – P; GMC1140
Dichanthelium oligosanthos (Schult.) Gould var. *scribnerianum* (Nash) Gould (Scribner's rosette grass) – P; GMC1101, GMC1152
Digitaria ciliaris (Retz.) Koeler (southern crabgrass) – A; GMC1230, GMC1255
Echinochloa muricata (P. Beauv.) Fernald (rough barnyardgrass) – A; GMC1264
 **Eleusine indica* (L.) Gaertn. (Indian goosegrass) – A; GMC1254
Elymus canadensis L. (Canada wildrye) – P; GMC1155
Elymus virginicus L. (Virginia wildrye) – P; GMC1210
 **Eragrostis cilianensis* (All.) Vign. ex Janchen (stinkgrass) – A; GMC1222
Eragrostis secundiflora J. Presl ssp. *oxylepis* (Torr.) S.D. Koch (red lovegrass) – P; GMC920
Eragrostis spectabilis (Pursh) Steud. (purple lovegrass) – P; GMC943
Erioneuron pilosum (Buckley) Nash (hairy woollygrass) – P; KR404
Hordeum pusillum Nutt. (little barley) – A; GMC1102, GMC791
 **Lolium perenne* L. (perennial ryegrass) – P; GMC788
Muhlenbergia racemosa (Michx.) Britton, Sterns & Poggenb. (marsh muhly) – P; GMC904
Panicum capillare L. (witchgrass) – A; GMC1218, GMC856
Panicum obtusum Kunth (vine mesquite) – P; GMC1248, GMC946
Panicum virgatum L. (switchgrass) – P; GMC874
Pascopyrum smithii (Rydb.) Á. Löve (western wheatgrass) – P; GMC1129
Phalaris caroliniana Walter (Carolina canarygrass) – A; GMC1083
 **Poa annua* L. (annual bluegrass) – A; GMC980
Poa arida Vasey (plains bluegrass) – P; GMC1018
 **Poa pratensis* L. (Kentucky bluegrass) – P; GMC990, GMC1007, GMC790, GMC1022
 **Schedonorus phoenix* (Scop.) Holub (tall fescue) – P; GMC1021
Sclerochloa dura (L.) P. Baeuv. (common hardgrass) – A; GMC977
Schizachyrium scoparium (Michx.) Nash (little bluestem) – P; GMC940
 **Secale cereale* L. (cereal rye) – A; GMC1011
 **Setaria pumila* (Poir.) Roem. & Schult. (yellow foxtail) – A; GMC1240
 **Setaria viridis* (L.) P. Beauv. (green bristlegrass) – A; GMC911, GMC1199
Sorghastrum nutans (L.) Nash (Indiangrass) – P; GMC898

**Sorghum halepense* (L.) Pers. (Johnsongrass) – P; GMC824, GMC912
Sporobolus compositus (Poir.) Merr. var. *compositus* (composite dropseed) – P; GMC931, GMC1223
Sporobolus cryptandrus (Torr.) A. Gray (sand dropseed) – P; GMC876, GMC1225, GMC1234
**Thinopyrum ponticum* (Podp.) Z.-W. Liu & R.-C. Wang (tall wheatgrass) – P; GMC1265
Tridens flavus (L.) Hitchc. (purpletop tridens) – P; GMC1213
Tridens muticus (Torr.) Nash var. *elongatus* (Buckley) Shinnery (slim tridens) – P; GMC 1224,
GMC1233
Tripsacum dactyloides (L.) L. (eastern gamagrass) – P; GMC847, GMC1184
Vulpia octoflora (Walter) Rydb. (sixweeks fescue) – A; GMC1033, GMC994



Figure 1 *Schizachyrium scoparium*-*Castilleja purpurea* var. *citrina*-*Lesquerella gordonii* herbaceous association on gypsum at Alabaster Caverns State Park. Photo courtesy of William Caire.



Figure 2 *Castilleja purpurea* var. *citrina* on gypsum outcrop at Alabaster Caverns State Park. Photo by G. Caddell.



Figure 3 *Lesquerella gordonii* with basal rosette of *Phacelia integrifolia* on gypsum outcrop at Alabaster Caverns State Park. Photo by G. Caddell.



Figure 4 *Phacelia integrifolia*, an obligate gypsophile, at Alabaster Caverns State Park. Photo by G. Caddell.



Figure 5 *Nama stevensii*, an obligate gypsophile, at Alabaster Caverns State Park. Photo by G. Caddell.



Figure 6 *Haploesthes greggii*, an obligate gypsophile, at Alabaster Caverns State Park. Photo by G. Caddell.



Figure 7 *Echinocereus reichenbachii* at Alabaster Caverns State Park. Photo by G. Caddell.

A COMPARISON OF THE COMPOSITION AND STRUCTURE OF TWO OAK FORESTS IN MARSHALL AND POTTAWATOMIE COUNTIES

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*Key words: Forest, composition, crosstimbers, science education,
vegetation structure*

ABSTRACT

In October 2011, high school students from McLoud High School sampled an oak forest in Earlsboro, Pottawatomie County. In July, 2012, students in the Pre-collegiate Field Studies Camp at the University of Oklahoma Biological Station sampled the Marshall County forest at the Buncombe Creek camp ground, located approximately 100 miles south of the Earlsboro forest and 1 mile north of the University of Oklahoma Biological Station. One component of each botany course was to study the composition and structure of an oak forest. These 2 forests were chosen to compare because of their similarity in composition and physical distance apart. They found 10 hardwood species in the Marshall County forest and 9 in the Pottawatomie County forest, with 6 species common to both. *Quercus stellata* was most important in both forests and most frequent in the Pottawatomie forest where the total density was 0.141/m². *Quercus stellata* and *Ulmus alata* were most frequent in the Marshall County forest where the total density was 0.107/m².

INTRODUCTION

The best way to learn how to identify the trees, shrubs, woody vines, and herbaceous plants of a forest, is to make frequent visits and practice field identification. High school students from McLoud High School and the Pre-collegiate Field Studies Camp at the University of Oklahoma Biological Station (UOBS) did just that; they made frequent visits, but to different forests. The McLoud High School students sampled a local forest, as well as a forest near Earlsboro, Oklahoma. After spending time in the forests, students learned to recognize the different shades of green, shapes and colors of tree bark, growth habits, blade complexity, leaf phyllotaxy, leaf margins, leaf shapes, leaf textures, leaf odors, and even the taste of leaves of different species.

By walking through the woods, I have learned the taste and effects of prickly ash—strong and bitter; numbing. I have learned the texture of hackberry leaves—scabrous and rough one way, smooth another. I have felt the barks of trees. All this I have learned by walking through the woods.

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In October, 2011, McLoud High School students studied an oak forest near Earlsboro in Central Pottawatomie County (35.425°, -97.0875°). In July, 2012, Pre-collegiate UOBS students studied an oak forest at the Buncombe Creek Camp Ground (33.52°, -96.48°), 100 miles south and 20 miles east of the Earlsboro forest, near the biological station in Marshall County. The two forests provide an

interesting comparison and contrast due to their similarity in composition and 100 mile north to south difference in location.

Students determined the composition of the forest by first learning to identify species within each of the quadrats. Students then collected data that can be used in long-term ecological studies. The structure of the forest was determined by calculating density, relative density, frequency, relative frequency, basal area, relative basal area, and importance values of those trees and shrubs in the forest. By measuring relative importance and frequencies of hardwood species, rather than calculating leaf area indices or other seasonal changes, their comparison of data taken in July in Marshall County to data taken in October in Pottawatomie County is still valid.

METHODS

Students set up eighteen 10 x 10 meter quadrats in each forest at a maximum distance from each other. This increased the likelihood of encountering a greater variety of habitats. In each quadrat, trees and shrubs were identified to species or genus, and then diameters of living woody stems 4 cm or greater at breast height (DBH) were measured. The more traditional method for measuring DBH has been to include stems 7.62 cm (3 in.) or greater (Greller et al. 1979, Phillippi et al. 1988, Rudnicki and McDonnell 1989, Stalter 1981). Including stems of 4 cm or greater will include more individual woody plants and yield a more complete data set than most traditional studies. A more recent study in New York (Glaeser 2006) measured DBH of woody plants that were 2 cm or greater. Measuring DBH at 4 cm or greater in this study may make direct comparisons with other studies using traditional measurements problematic, but a more accurate comparison of these 2 sets of forest data is possible. With the number of student data collectors in a field class and the use of computers which can

handle greater sets of data, this can be a cost-effective way to improve data collection for long-term studies.

Students were taught to determine density, relative density, frequency, relative frequency, basal area, and relative basal area for individual species using a simple calculator. To save time and improve accuracy, data from the forests were entered in an Excel 2010 program for 18 quadrats from each forest. Importance values were calculated by adding the three relative values for each species

RESULTS

In the Marshall County forest, 10 species were identified in the 1800 m² sampling area. In the Pottawatomie forest, 9 species were found in the 1800 m² sampling area. The 2 forests had 6 species in common: *Quercus stellata* (post oak), *Q. marilandica* (black jack oak), *Carya texana* black hickory, *Fraxinus americana* (white ash), *Ulmus alata* (winged elm), and *Juniperus virginiana* (eastern redcedar).

U. alata had the highest density in the Marshall County forest. *Q. stellata* had the highest density in the Pottawatomie forest. *Q. stellata* and *U. alata* had the highest frequency in the Marshall County forest. *Q. stellata* had a frequency of 1.00, the highest frequency in the Pottawatomie forest. *Q. stellata* had the highest basal area in both forests. The 2 trees with the highest importance values respectively in both forests were *Q. stellata* and *U. alata*. The total density for the Pottawatomie forest was 0.141 trees/m² and the Marshall County forest was 0.107 trees/m². The total basal area for the Pottawatomie County was 21.2 cm²/m². The total basal area for the Marshall County forest was 23.3 cm²/m². The 6 common species in both forests had a relative importance of 0.944 for the Marshall County forest and 0.954 for the Pottawatomie County.



Figure 1 Buncombe Creek Forest, Marshall County, Oklahoma

Table 1 Density, frequency, basal area, and importance values for the Buncombe Creek Forest, Marshall County.

Species	Density, trees/m ²	Frequency	Basal area cm ² /m ²	Importance value
<i>Quercus stellate</i>	0.0233	0.944	14.3	1.06
<i>Ulmus alata</i>	0.0422	0.944	2.14	0.710
<i>Juniperus virginiana</i>	0.0161	0.722	1.31	0.378
<i>Quercus marilandica</i>	0.0106	0.500	2.82	0.338
<i>Carya texana</i>	0.00222	0.222	0.751	0.106
<i>Fraxinus americana</i>	0.00778	0.444	1.42	0.239
<i>Morus rubra</i>	0.00222	0.222	0.0850	0.0768
<i>Vaccinium</i> spp.	0.000556	0.0556	0.00698	0.0186
<i>Prunus mexicana</i>	0.000556	0.0556	0.00698	0.0187
<i>Quercus velutina</i>	0.00111	0.111	0.461	0.0565



Figure 2 Earlsboro Forest, Central Pottawatomie County, Oklahoma

Table 2 Density, frequency, basal area, and importance values for the Earlsboro forest, Pottawatomie County.

Species	Density, trees/m ²	Frequency	Basal area cm ² /m ²	Importance value
<i>Quercus stellata</i>	0.111	1.00	18.9	2.05
<i>Ulmus alata</i>	0.0183	0.667	1.56	0.454
<i>Juniperus virginiana</i>	0.00278	0.222	0.0938	0.108
<i>Quercus marilandica</i>	0.00278	0.222	0.233	0.114
<i>Carya texana</i>	0.000556	0.0556	0.0109	0.0253
<i>Fraxinus americana</i>	0.00222	0.222	0.241	0.110
<i>Amelanchier spp.</i>	0.00111	0.111	0.0650	0.0526
<i>Celtis spp.</i>	0.00167	0.111	0.0785	0.0572
<i>Quercus shumardii</i>	0.000556	0.0556	0.0279	0.0261

DISCUSSION

The relative importance values for the 6 common species show 2 very similar forests even though they are separated by at least 100 miles. At the same time, they are very different in terms of their composition of shrubs, understory trees, vines, and herbaceous plants of the forest floor. The Marshall County forest has a much denser forest floor, understory layer, and shrub layer than does the Pottawatomie County forest (Figures 1 and 2). Another major difference in the 2 forests is the dominance of post oak in the Pottawatomie forest, where *Quercus stellata* had the highest density, frequency, basal area, and importance value. The density of post oaks in the Pottawatomie forest is almost five times greater and the importance value is nearly two times greater than the post oaks in the Marshall County forest even though the post oak basal area did not differ much. Future studies might reveal the cause for these differences.

As a part of a field learning experience, students are able to collect large data sets over a long period of time, which might otherwise be prohibitively expensive to obtain. Furthermore, getting students into the field provides them with a depth of knowledge they could not possibly learn from reading a text or looking at dried specimens. While these studies provided an opportunity to begin a long-term ecological research project that involved students in field research, student identification of species in the field could be inaccurate to the point that it renders data useless. However, we found that allowing students time in the field to learn species identification (using more than a key and dried specimens) before beginning the field study, appeared to increase their accuracy. Students received immediate feedback regarding the accuracy of their species identification from instructors and teaching assistants, who were in the field with them.

The ecological value of this student research is that it creates baseline data for further research, to track changes in the 2 forests with possible links to changes in species due to global climate change. The greater value of this research is the invaluable experience for high school students, increasing their knowledge of nature and science aptitude by actually being in the natural environment (Louv 2011). They learn more than facts. They learn *how to learn* from the forest.

*As I was walking through the forest; sun
shining, elm leaves fluttering, birds flying,
critters bustling, it occurred to me; mother
nature teaches the purest kind of wisdom:
you don't need to be in a classroom to
learn. Knowledge is everywhere.*

*Magen Clark and Caitlyn Carr
McLoud High School
McLoud, Oklahoma*

Beginning this long term study will also provide a beginning set of data to test hypotheses regarding how students learn in the field, versus how they learn in the lab or classroom. While I am confident that students have learned to identify trees during this project, future field studies should be accompanied by assessment of student identification skills comparing both field and laboratory experiences. This outdoor experience meets C3 PASS Standards 1 and 2 (Oklahoma PASS 2006) for general biology.

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Critic's Choice Essay

VIRTUAL HERBARIA COME OF AGE

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These are exciting times for natural history collections. An international effort is underway to make images and data of biological specimens available in electronic format via digitization. These initiatives are an effort to bring natural history collections *out of the dark* of museum and herbarium cabinets and *into the light* of public access for use by stakeholders in government, academia, biodiversity organizations, business, and K-12 education. The democratization of information contained in natural history collections through images and online databases is an important new development to better investigate our natural world and solve important social and environmental problems (Scoble 2010).

For herbarium collections, digitized images and data from specimens are generally referred to as a virtual herbarium. What exactly do we mean by digitization of natural history collections? For plants, digitizing collections transforms herbarium specimens into digital images and label data sorted (parsed) into its component units such as names, locations, collectors, dates, habitats, and reproductive state. All data and images are fully searchable and distributed in electronic format, such as virtual herbaria. There are several outstanding examples of virtual herbaria already online, such as Australia's Virtual Herbarium (<http://avh.ala.org.au/>) and the New York Botanical Garden's Virtual Herbarium (<http://sciweb.nybg.org/science2/VirtualHerbarium.asp.html>). According to the US Interagency Working Group on Scientific Collections (IWGSC 2007), plant specimen

data distributed via virtual herbaria would have a profound impact on science education and investigations of environmental change and quality, invasive species, public health, national security, bioscience research, and many other issues (NIBA 2010).

Why digitize natural history collections? Think for a moment about the incomparable treasure trove of biodiversity information contained in the world's natural history collections. If we focus just on plants, herbarium specimens document most of what is known about the world's plant species diversity and represent a 200+ year record of what species were present at a given *location* and at a given *time*. Herbaria collections not only document the different kinds of plants constituting a flora, but they record valuable information about where they occurred and when they were flowering or fruiting. Plant specimens provide a spatial and temporal window into the dynamic processes of plant diversity, introduction and spread of exotics, expansion and contraction of species ranges, and changes in time of flowering and fruiting. Is a digitized herbarium specimen as valuable as the specimen itself? Certainly not, and this is one of the main reasons for maintaining natural history collections in museums and herbaria. The primary rationale for digitizing specimens is **access**. Scientists throughout the world will have greatly enhanced access to digitized specimens, which greatly adds to their value for research and education.

In the US, a key component is in place to assist efforts to digitize biological research collections (e.g., herbarium specimens) – the Integrated Digitized BioCollections resource

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(iDigBio; www.idigbio.org). Tools and training provided by iDigBio are funded by the National Science Foundation, who also established a 10-year funding program entitled Advancing Digitization of Biological Collections (ADBC) to aid conversion of biodiversity collections into electronic formats. These advances in funding and infrastructure were established using recommendations of the National Science and Technology Council, who recognized the importance of biocollections for national science infrastructure.

With an estimated 90 million herbarium specimens in U.S. herbaria (Tulig et al. 2012), is it feasible to construct a US Virtual Herbarium comparable to Australia's Virtual Herbarium based on "only" 6 million specimens? At a minimum, digitization of biocollections involves specimen imaging, image processing, electronic data capture, and georeferencing of locality descriptions (Nelson et al. 2012). Mass digitization methods continue to be refined and automated (Beaman and Cellinese 2012), but it is unlikely that all US herbarium specimens can be digitized in a 10-year timeframe. However, a recent survey conducted by the US Virtual Herbarium project (Barkworth and Murrell 2012) indicated that ca. 30% of herbarium specimen labels were already databased. While it appears that much digitization has occurred at the individual herbarium or regional level, there has been no coordinated national effort to expedite digitization of biocollections. However, the iDigBio mission aims to fill that void and has a major objective to facilitate access to US biocollection data. This goal is certainly feasible, especially since a **global** portal for digitized images and data from natural history collections already exists – GBIF, the Global Biodiversity Information Facility (www.gbif.org). GBIF currently serves up more than 300 million specimen records.

Herbaria throughout the country are actively engaged in efforts to image and database information and to present them in

searchable online formats. International standards and best practices for data capture have been established and are being implemented by the collections community nationwide. Luckily, botanists in Oklahoma demonstrated great foresight by establishing a data portal for digitized herbarium label data for specimens collected in Oklahoma – the Oklahoma Vascular Plants Database (OVPD; Hoagland et al. 2004). With the OVPD as a firm foundation and a collaborative network in place among curators in the state and region, Oklahoma herbaria are poised to expand their digitization efforts. This endeavor will help develop the concept of a virtual herbarium to maturity and will undoubtedly enhance the value and access of real herbaria.

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