International Journal of Agriculture Sciences
ISSN: 0975-3710 & E-ISSN: 0975-9107, Volume 15, Issue 6, 2023, pp.-12396-12402.
Available online at https://bioinfopublication.org/pages/jouarchive.php?id=BPJ0000217

Review Article

WEED SEED BANK AND ITS SIGNIFICANCE IN WEED MANAGEMENT

BARLA S. AND UPASANI R.R.*
Department of Agronomy, Birsa Agricultural University, Ranchi, 834006, Jharkhand, India
*Corresponding Author: Email - upasani.ravikant@gmail.com

Received: May 07, 2023; Revised: June 26, 2023; Accepted: June 28, 2023; Published: June 30, 2023

Abstract: The store of healthy weed seeds that are dispersed across the soil profile and on the soil's surface is known as the weed seed bank. The creation of integrated weed management (IWM) programs can benefit from an understanding of the elements influencing the dynamics of weed seed banks because agricultural soils can have thousands of weed seeds per unit square area. Management should incorporate tactics to reduce the weed seed bank in addition to agricultural yield loss considerations. Numerous interrelated factors, including production practices and environmental circumstances, affect the persistence of viable seeds in the soil seed bank. Seed dormancy, physiological age, predation, microbiological decomposition, environmental circumstances, burial depth, burial time, and tillage are a few of the factors affecting weed seed bank. For a thorough understanding of the weed seed bank, it is crucial to have a solid grasp of weed biology, including morphology, lifecycles, seed generation.

Keywords: *Germination, Seed predation, Weed dormancy, Management, Weed seed bank*

Citation: Barla S. and Upasani R.R. (2023) Weed Seed Bank and Its Significance in Weed Management. International Journal of Agriculture Sciences, ISSN: 0975-3710 & E-ISSN: 0975-9107, Volume 15, Issue 6, pp.- 12396-12402.

Introduction

The seed bank is a crucial part of the life cycle of weeds because it serves as the last resting place for weed seeds. Future weed populations of annual and perennial weed species that only reproduce by seeds can be found only in seed banks. Understanding the destiny of seeds in the seed bank can therefore be a crucial part of weed control in general. When weed seeds reach the seed bank, a number of factors affect how long the seeds will persist. In the seed bank, seeds are able to sense their immediate surroundings and use this information to either go dormant or start germination. In order to control the longevity and germination characteristics of weed seeds, soil and crop management measures can have a direct impact on the environment of seeds in the soil weed seed bank. Weeds are regarded as one of the main limiting factors in crop output among the other causes. It is therefore, crucial to control weeds at the lowest possible cost and with the least amount of environmental damage. Hence, for better understanding the significance of weed seed bank and its management for sustainable crop production an effort has been done in this chapter to review work done by several workers on similar issue.

Weed Seeds

Weed seeds are defined as the seeds of all plants that are commonly regarded as weeds and includes noxious weed seeds that are prohibited and restricted. Since most weeds in agricultural areas multiply and survive as seeds, the soil weed seed bank serves as the primary source of weed infestations in the future. Despite efforts to eradicate them, weeds often continue to proliferate, and their ability to persist depends on the soil seed bank [1]. Soil seed banks act as genetic resource banks that provide a variety of responses to environmental conditions and protect populations against transiently unfavorable environmental situations [2]. The soil seed bank controls many weed communities. Soil seed bank dynamics must be understood to create more effective weed management strategies [3]. According to Forcella *et al*. (1993) [4], a thorough understanding of weed ecology, including seed bank density, seed dormancy, seedling emergence, and environmental variables that govern these characteristics, is required to reduce the chemical pesticide burden on the environment without reducing crop output.

It comprises both freshly shed weed seeds and older seeds from prior years that have remained in the soil [5]. In addition to providing a physical record of past cropping system triumphs and failures, the weed seed bank can assist farmers in estimating the extent to which crop-weed competition will affect crop output and quality. The creation of integrated weed management (IWM) programs can benefit from an understanding of the elements influencing the dynamics of weed seed banks because agricultural soils can have thousands of weed seeds per unit square area. Management should incorporate tactics to reduce the weed seed bank in addition to agricultural yield loss considerations. According to Cousens and Mortimer (1995) [6], persistent weed infestations in agricultural fields are mostly caused by weed seed banks, and if their deposits rise, more herbicide dosages will be needed to manage weeds later [7].

In contrast to perennials, which can spread through seed production as well [8], annual weed species only increase their populations through seed production [9]. Knowledge of the weed seed bank's contents (size and species composition) can assist producers to foresee and mitigate the effects of crop-weed competition on crop yield and quality. The weed seed bank serves as a tangible chronicle of the past successes and failures of cropping systems.

Type of Weed Seed Bank

According to Thompson, and Grime, (1979) [10], a seed bank might be either transient or persistent, in general. According to the definition of a transient seed bank, seeds do not survive until the second germination season after maturation, whereas seeds in a persistent seed bank do survive till the second or future germination season [11].

Weed scientists made a distinction between transient seed banks, which are present for less than a year and include, for example, grass seed, short-term persistent seed banks, which are present for at least a year but less than five years, and long-term persistent seed banks, which are present for at least five years [12]. The persistent seed bank is of greater interest to weed scientists than the transient seed bank. Future weed management strategies should be concerned with weed species that produce persistent seed banks.

Factors affecting weed Seed Bank in Soils

Numerous interrelated factors, including production practices and environmental circumstances, affect the persistence of viable seeds in the soil seed bank. Seed dormancy, physiological age, predation, microbiological decomposition, environmental circumstances, burial depth, burial time, tillage and weed species are a few of the factors affecting weed seed bank [13].

Dormancy of Weed Seeds

Dormancy of weed seeds is a very crucial feature of weed seeds as it ensures for most weed species only a small proportion of buried weed seeds is germinated as seedlings from the soil seed bank in any given year [14]. Physical dormancy is caused by an impenetrable seed coat to gases and water, while physiological dormancy is caused by hormones, phytochromes, and inhibitors, and morphological dormancy is caused by an underdeveloped embryo. According to a study by Roberts and Feast (1973) [15], the weed seed bank decreases by 32% every year in areas with temperate climates. In contrast, the weed seed bank is typically smaller and the decline generally occurs more quickly in tropical regions due to (a) a high seed germination rate favoured by conducive climate conditions which persist for longer periods of time than in temperate regions; (b) a high seed mortality due to attack by predators; (c) high relative humidity and higher temperatures, which favour biotic agents;(d) reduced seed viability; (e) a shorter duration of seed dormancy in many weed species; and (f) seedling mortality owing to seed germination in brief, hot, dry spells that can occur throughout the rainy season.

Physiological age of Weed seed:

Gibberellins, ethylene, cytokinins, or abscisic acid (ABA) all play a significant role in inducing or suppressing seed dormancy from a physiological standpoint. In seeds of several plants, such as *Ricinus communis, Lactuca sativa*, and *Hordeum vulgare*, as well as weed species, such as *Avena fatua*, the low level of ethylene is accumulated at the early stage of germination shortly before radical protrusion. *Xanthium pensylvanicum* embryos that are not dormant generate more ethylene than dormant seeds [16]. During the phases of dormancy breaking and freezing, the levels of cytokinins in the dormant seeds of *Rumex obtusifolius* and *Spergula arvensis* rose. Inducing dormancy or promoting seed germination and dormancy breaking include ecological conditions. Light, temperature, O_2, CO_2, and nitrate are a few of these elements. Weed seed dormancy is brought on by light. Some weed seeds, such as those of *Galinsoga parviflora* Cav., *Portulaca oleracea* L., Chenopodium album, and Amaranthus spp., need light to germinate. Light, which comes in both promoting and inhibitory forms, is associated with breaking dormancy. Red light is preferred for promoting form and far-red light for preventing form. When a pigment absorbs far-red light around 750 nm, germination typically begins, however when pigment absorbs red light around 660 nm, germination is either prevented or unaffected.

However, dormancy can be enhanced or broken by the physiological actions of light, moisture, temperature, and oxygen. Water or moisture is necessary to activate enzymes, replenish water lost by the developing embryo during respiration, and dissolve and move food into the developing body. While water absorption, hormonal balances, metabolic processes, and germination induction will not occur unless a specific appropriate temperature is reached, aerobic respiration, which uses oxygen to supply energy for embryo growth, will not be possible without oxygen. However, every element is necessary for all biochemical and physiological processes, including the development of the living embryo, to take place inside the seed.

Predation

One of the most crucial strategies for eliminating weed seeds from agricultural ecosystems is seed predation. Predation on weed seeds occurs in the form of ground beetles, crickets, earthworms, slugs, field mice, and birds, weed seeds are no different from the seeds of most plant species in that they can be nutritious food for birds, insects, and other living things. Weed seeds are consumed by insects and other creatures both while they are still attached to the parent plant and after they have been shed. Pre-dispersal predation on velvetleaf, lambsquarters, pigweeds, and Canada thistle by specialized insects (species that consume just one type of weed seed) has been observed by White *et al*., (2007) [17]. Annual weeds may be suppressed in crop fields by predators that use weed seeds as a valuable food source [18]. Researchers have demonstrated that controlling weeds is a critical issue in all farming systems to enable predators to lower weed seed density. For instance, over a 2-week assay period conducted in conjunction with natural seed rain, Mauchline *et al*. (2005) [19] discovered greater than 70% predation of common chickweed (*Stellaria media* (L.) Vill.], Prostrate knotweed (*Polygonum aviculare* L.), common lambsquarters (*Chenopodium album* L.), and wild mustard (*Sinapis arvensis* L.). According to research investigations by Zhang *et al*. (1997) [20], reports of 90% or more seed loss or damage are common for some weed species. Weed seed predators cannot get rid of all weed seeds, especially those from weed species with large seed production. However, researchers have shown that over time, weed seed banks can be depleted if weed control strategies are not planned successfully to prevent adding more seeds to seed banks each year. Weed species with low seed densities may experience a sharp drop in their seed banks and population dynamics over successive seasons as a result of seed predation. Weed seed banks are more widely affected by post-distribution weed seed predation. Millions of weed seeds are annually removed from farmers' fields by a cleanup team comprising insects, other invertebrates, small rodents, and seed-eating birds, particularly during and in the weeks after weed seed is shed. Ground beetles (carabids) are among the most extensively studied and consistent weed seed consumers [21]. Many weed seed consumers appear to be sufficiently selective to qualify as primarily helpful creatures unlikely to harm crop establishment or yield, even if others will take crop seeds along with weed seeds. Numerous weed seeds have been demonstrated to be consumed by ground beetles, crickets, and even white-footed field mice without having a substantial negative influence on freshly seeded crops. Crop seeds are frequently planted deeply enough to avoid predation, but these seed-eaters mostly eat seeds that are on or very near the soil surface. Smaller ground beetles that devour annual grasses, pigweed, and other small weed seeds lack the physical strength to ingest larger crop seeds. Crows, blackbirds, and several other seed-eating birds can severely harm newly-seeded corn and other crops, while slugs can gravely harm sensitive young vegetable crops while also consuming weed seeds [22].

Microbiological Decomposition

When seeds detect signals from dangerous bacteria, they stop germination to prevent infecting seedlings. As long as the environment is unfavorable, the seeds can prevent germination. Limited research suggests that microbes connected to weed seeds can contribute to the depletion of seed banks by chemotaxis (attraction to seeds), fast spermosphere colonization, and synthesis of enzymes and/or phytotoxins to destroy seeds before germination. Future weed control strategies aimed at seed banks will be significantly impacted by a fundamental understanding of interactions between seeds and microbes. According to Joanne *et al*. (2006) [23] in laboratory studies, the susceptibility of various weed species to seed decay ranges from high (velvetleaf) to very low (giant ragweed). Microscopic investigations showed that anytime seeds were exposed to soil microorganisms, dense microbial assemblages developed, regardless of whether the result was degradation. Pseudomonas aeruginosa is a bacteria that can be harmful to both plants and animals, including humans, according to research done at the University of Geneva (UNIGE), in Switzerland. Jean-Luc *et al*. (2018) [24], have found that this bacterium produces a toxin known as AMB that causes the seeds to delay germination without harming the plant. Therefore, it is possible that throughout evolution, the seeds developed the ability to defend their species by using AMB, a signal sent by bacteria to coordinate their infection strategies.

Environmental circumstances

Predicting the emergence of weed seedlings requires knowledge of how the dominant weed species in farmland germinate their seeds. To make better weed management decisions, the ability to predict seedling emergence is crucial. The emergence of weeds, however, is the consequence of two independent processes, namely germination and pre-emergence growth of shoots and roots, which respond differentially to environmental conditions.

Soil Temperature

Soil temperature is arguably the most pronounced and noticeable factor influencing emergence in temperate areas [25]. In crop growth models, soil temperature can be utilized as a predictor of seedling emergence (Angus *et al*., 1981). If emergence can be predicted by a straightforward continuous cumulative sigmoidal curve and the top few centimeters of soil are consistently moist, soil temperature can also be used to forecast the appearance of weeds. It has been noted by Gardarin *et al*., (2010) [26] that appearance of weeds depends on the rate and timing of seed germination, which is influenced by moisture potential as well as soil temperature. Soil temperature is one of the main environmental elements that regulate seed behavior in the field. It controls seed dormancy, which affects the germination capacity, as well as the rate or speed of germination in non-dormant seeds [27]. According to Colbach *et al*. (2002a) [28], the pre-emergence growth of shoots and roots should be researched and modeled independently since they respond to environmental stimuli differently. The rate of seed germination also varies with temperature, increasing in the suboptimal range and decreasing above the optimum temperature. Varying weed species require different minimum temperatures for seed germination. According to Colbach *et al*. (2002b) [29], the minimum temperature needed for seed germination is 0°C for both the summer annual *P. aviculare* and the winter annual *A. myosuroides* [30]. Masin *et al*. (2005) [31] calculated the base temperatures for Eleusine indica (L.), *Digitaria sanguinalis* (L.), *Setaria viridis* (L.), *P. Beauv., Setaria pumila* (Poir.), *Roem. & Schultes, and Setaria pumila* (Poir.), respectively, at 8.4, 6.1, 8.3, and 12.6°C. Additionally, the mean Tb (base Temperature) for the summer annuals *Amaranthus albus* (L), *Amaranthus palmeri* (S. Wats.), *D. sanguinalis, Echinochloa crus-galli* (L.) Beauv., *Portulaca oleracea* (L.), and *Setaria glauca* (L) was approximate ~40% higher than the corresponding value for the winter annuals *Hirschfeldia incana* (L) and *Sonchus o* (L.). Batlla and Benech-Arnold, 2005 reported that the optimal temperature range needed to end dormancy varies depending on the species. *Panicum miliaceum* (L.) seeds, for instance, shed their dormancy at 8°C, but *P. aviculare* seeds did so at 17°C. Benech-Arnold *et al*., (2000) [32] opined that once the degree of dormancy is sufficiently low, fluctuating temperatures are among the factors that can eliminate restrictions on the germination of many weed species' seeds. Particularly, the magnitude and frequency of diurnal soil temperature changes may play a significant role in reducing the dormancy of some species' seeds. As an illustration, compared to non-alternating temperatures, germination of *Amaranthus retroflexus* (L.), *Amaranthus spinosus* (L.), and *Amaranthus tuberculatus* (L.) rose from 23 to 65, 8 to 77, and 9 to 57%, respectively. However, there is a need for more research on how noxious weed species in various parts of the world and under diverse soil and climatic circumstances respond to temperature fluctuations because different weed species have varying requirements for variable temperatures for seed development.

Soil Moisture

Evans and Etherington, (1990) [33] suggested that the soil moisture status in wild plants' natural environments may be related to how their seeds germinate in response to soil water potential. Varying weed species' seeds require different levels of water potential for germination. According to Colbach *et al*. (2002b) in a study, the base water potential (Ψb) for *A. myosuroides*, for example, was calculated to be 1.53 (MPa), although other researchers found that the similar value for *Ambrosia artemisiifolia* (L.) was 0.8 (MPa) [34]. *Stellaria media* (L.) Villars had a corresponding value of 1.13 (MPa), whereas the lowest water potential for the germination of *S. viridis* seeds was 0.7 (MPa) [35]. According to Dorsainvil *et al*. (2005) [36], *Sinapis alba* (L.) had a basal water potential for germination of 1. (MPa). Singh and Singh (2009) [37] experimented to see the effect of temperature and water potential on the germination of test species namely *Brazil pusley*, common ragweed, Florida beggarweed, hairy beggarticks, ivy leaf morning glory, Johnsongrass, prickly sida, redroot pigweed, sicklepod, strangler vine, tall morning glory and yellow nutsedge). According to them the germination of weed species was reduced by 12, 32, 75, and 96%, respectively, when water potential was reduced from -0.1 to -0.2, -0.4, and -0.8 MPa when data were averaged across species. According to them no germination was seen in none of the test species, at -1.2 MPa osmotic potential. Compared to common ragweed, *Brazil pusley*, strangler vine, and redroot pigweed, Florida beggarweed and tall morning glory were less affected by decreasing water potential during germination. They further reported that at -0.1 MPa osmotic potential, increasing the temperature from 15 to 20 and 30°C led to germination rates of 18, 36, and 56%, respectively. Many weed species were more inhibited by low temperatures and reduced water stress than by high temperatures and enhanced water stress. All species had their germination completely inhibited by a decrease in osmotic potential of -1.0 MPa, except Florida beggarweed (11%). Tall morning glory's germination rose from 10 to 34 and 43% at -0.5 MPa osmotic potential with temperature increases of 5 or 10°C. Florida beggarweed did not germinate at -0.5 MPa at 15 or 20°C compared to 43% at 30°C, which was comparable to 20°C and had less water stress (-0.1 MPa).

Thill (1979) [38], studied the effects of soil moisture, temperature, and compaction on the germination and emergence of downy brome (Bromus tectorum), found that lowering the soil matric potential from -2 to -16 bars significantly reduced the proportion and rate of weed seed emergence. Temperature consistency was preferable to changing temperatures for seedling emergence. Warmer soil temperatures (20°C) enhanced emergence rates at high matric potentials, while at very low matric potentials, cooler temperatures had the least impact on emergence rates and percentages (10 and 15°C). Katsuyoshi and Kanenori (1979) [39] found that seeds' capacity to absorb moisture was high in crops but low in weeds and grasses. In high osmotic pressure conditions, plant development was inhibited. They further reported that 3) After several days of irrigation in the fields, the soil moisture percentage at the ground's surface (0–1 cm) dropped to 25–30%, which was below the minimal soil moisture levels required for weed emergence. However, the soil moisture in the bottom layer (1-2 cm) was very consistent, making it obvious that a slight variation in the placement of the seeds was crucial for their emergence.

Effect of Tillage and burial depth of weed seeds

The nature and growth of weed species are greatly impacted by tillage. Different tillage techniques are practiced globally, with conventional tillage accounting for the majority and conservation tillage or reduced tillage has come into practice recently. The reason why or how tillage affects weed seed banks is that the weed seeds build up at or near the soil surface when soils are not periodically disturbed by ploughing. In general, the majority of annual weeds may easily germinate and emerge from soil depths of 0 to 5 cm. The plants that grow from those seeds have a harder time emerging when they are buried further in the soil profile by a soil inverting plough. There is no doubt that seed size matters and plants with larger seeds have a better chance of emerging from deeper burial depths than those with smaller seeds (more energy in bigger seeds).

Sharma *et al*. (2004) [40] opined that conventional tillage promotes more monocot weed infestation while zero tillage (ZT), on the whole, promotes the establishment of dicot weeds. Clements et.al. (2017) [41] studied the impact of four tillage systems by moldboard plow, chisel plow, ridge-till, and no-till on weed seed return and seed bank composition under corn-soybean crop rotation. In chisel plough and no-till methods, they concluded that more than 60% of the weed seed bank was localized in the top 5 cm of the soil. In comparison to the other methods, the moldboard plough system's seed bank was larger and more evenly dispersed throughout the depth. In all systems besides ridge-till, common lambsquarters made up more than 50% of the seed bank, yet they only predominated the aboveground weed population in the chisel plough system. Moldboard ploughing increased common lambsquarter seed bank populations more than ridge-till, no-till, and chisel plough, which increased seed bank populations more than ridge-till. Common lambsquarters were more prevalent aboveground in chisel and moldboard plough methods than in the other two. Common lambsquarters had equal seed output per plant in all four systems, however, moldboard plough and chisel plough systems had higher estimated seed production per unit area than the other systems. In moldboard plough and chisel plough systems, populations of common lambsquarters and related species may produce more seeds and persist; in no-till and ridge-till systems, these weeds may produce fewer seeds per unit area and be simpler to control.

Buhler (1995) [42] reported that if primary tillage is employed in conjunction with delayed planting, which enables the annual species to sprout before the tillage operation, annual weed management can be significantly improved. The effectiveness of tillage as a control strategy, however, can be diminished if it is postponed until weeds grow larger (Hager, 2013) [43]. Herbicide treatment later in the season may be more challenging for summer annual weeds that are not eliminated by tillage [44]. The moldboard plough and chisel plough are two examples of primary tillage tools, with the moldboard plough being more effective at burying weeds and their seeds. Ona Aukalnien (2018) [45] investigated the impact of different tillage practices, including conventional tillage (CT), moldboard ploughing at 22–24 cm depth, minimum tillage (MT), and stubble cultivation at 10–12 cm depth, on weed counts and found that the no-tillage plots had the highest number of weed seeds per square meter (14700). While the seed counts in the conventional tillage plots were much lower, similar seed counts (14233) were recorded in the minimum tillage plots (over five years the soil weed seed bank significantly decreased). They also claimed that, in comparison to traditional tillage, the number of weed seeds was much higher in less disturbed soil.

Effect of Cropping system and Crop Rotation

The dynamics of weed populations in arable fields are also influenced by the cropping system and their management and crop rotations are altered. For instance, annual grass weeds were more prevalent in fields of continuous winter wheat, but broadleaf weeds were more prevalent in areas of sugar beet-winter wheat cycle. Koocheki *et al*. (2009) [46] studied the effect of different cropping systems and crop rotations on the weed population and seed bank. They conducted a trial in a split-plot design with three various crop rotations in main plots and five different cropping systems consisting of high-input, medium-input, low-input, organic, and integrated systems applied to the sub-plots. The objective was to find whether crop management ranging from low- to high-input could change weed seed bank characteristics over time. According to their findings, weed seed densities in organic and integrated cropping systems were higher than those in conventional and high-input cropping systems, which had roughly 2000 seeds per square meter. Compared to other rotations, which had about 5000 seeds/m², continuous winter wheat showed a higher density of weed seeds. In the high-input system, there were 11 types of weeds and 66 plants per square meter. The weed populations in the organic and low-input systems were 15 and 13 species, with 145 and 220 plants/m² respectively. They also reported that organic and integrated cropping systems had a higher proportion of seeds in the top 15 cm of soil than the lower layer (15–30 cm) and that high-, medium-, and low-input systems had a much more uniform distribution along the profile (0–30 cm). This outcome was brought about by these systems' use of conventional tillage (moldboard tillage). According to Buhler *et al*. (2001) [47], the tillage system affects how weed seeds are distributed in the soil. They found that normal tillage evenly distributes seed among different soil aggregate classes in the soil's top layer, but reduced tillage tends to concentrate weed seeds in the un - aggregated soil fraction. Finally, they concluded that organic and low-input systems were more weed-infested. The long and diverse crop rotations with careful weed control in low-input and organic systems are fundamental in sustainable and ecological crop production systems. Studying the effect of cover farming on weed seed banks, Nicholus *et al*. (2020) [48] found that, under some situations, cover cropping can significantly lower the size of the weed seed bank compared to a no-cover control. They also emphasized the need for greater study in long-term plots comparing cover crop effects in different cropping systems and management regimes to pinpoint the situations where cover crops are most successful at preventing and/or reducing weed seed deposition.

Weed species

Future weed management strategies should be concerned with weed species that produce persistent seed banks. It has been reported by several workers that although genetically modified (GM) herbicide-resistant crops are widely used and herbicide-resistant (HR) weed biotypes have developed, GM farming systems have had very little impact on weed species communities [49,50]. Walsh *et al*. (2013) [50]also reported that Simulation models for herbicide resistance have unequivocally shown that the risk of resistance is inversely correlated with the quantity of the soil seed bank.

The most important aspect in determining the success of subsequent generations of weed species is how long seeds remain in the soil. Burnside *et al*.(1996) [51] reported that long periods of burial have been known to maintain some weed species' viability and persistence. For instance, morning glories (*Ipomoea* spp.) can survive in the soil seed bank for at least 39 years [52]. Some weed species, such as common cocklebur (*Xanthium strumarium* L.), *Sisybrium orientale* L., prickly sida (*Sida spinosa* L.), spurges (*Euphorbia* spp.), water hemp (*Amaranthus tuberculatus*), and redroot pigweed (*Amaranthus retroflexus* L.), will approach exhaustion by 3 to 4 years in the absence of seed return, The majority of weed species, however, lose their viability as seeds after only a brief period of burial.

According to studies by Roberts and Neilson(1981) [53], if little to no seed is put back into the ground, the seed bank can be decreased by at least 90% within four years. Furthermore, according to Burnside *et al*. (1986) [54] during a 5-year experimental period, soil weed seed bank decreased by 95% when weed seed return was blocked (20 seed to 1 seed 454 g−1 soil). However, they further observed that at two of the five locations, one year without weed control and five years without any weed seeds returning to the soil seed bank was sufficient to refill the weed seed bank population. This demonstrates the need for an efficient weed management program to maintain low soil seed bank levels. The target weed species must be managed in a way that minimizes seed return to achieve this level of reduction.

Distribution of Weed seeds in the soil profile

In the soil profile, weed seeds can move both horizontally and vertically. While the direction of crop rows generally determines the horizontal distribution of weed seeds in the seed bank, the primary determinant of the vertical distribution of weed seeds within the soil profile is type of tillage. In fields that have been ploughed, the bulk of weed seeds are buried four to six inches below the surface. A good 80 to 90 percent of weed seeds are dispersed in the top four inches of the soil profile when using reduced tillage techniques like chisel ploughing. The bulk of weed seeds remain at or close to the soil surface in no-till areas. There is evidence that soil properties affect the vertical dispersal of weed seeds even though very few studies have examined the impact of tillage practices on various soil types [55]. Weed seeds typically remain on the soil surface in cropping systems without soil disturbance and tillage, as is the case for subsistence farming, where they are easier to manage ([56].

Weed seed bank research methodology

According to Forcella *et al*., (2003), there are two methods to enumerate the number of seeds in the soil i.e. direct seed extraction and germination method:

Direct seed extraction method

This method involves the separation of weeds from the soil by washing or flotation. On a screen with a mesh size smaller than the smallest anticipated seed, the soil sample is initially laid out. The majority of tiny seeds may be caught with a mesh size of roughly 0.2 mm. The seeds must be recognized after being separated using the direct seed extraction method. Under magnification, identification is accomplished utilizing appropriate literature.

Germination method

The density of dormant seeds in the seed bank is counted using this method. Twenty cores are advised following an experimental procedure. The cores are combined, assembled, put in trays, and then put in a greenhouse. To ensure that all seeds can germinate, the ideal soil depth in the trays should be between 2-3 cm, with a maximum of 5 cm. To allow for drainage, trays ought to be perforated.

Research on seed banks has recently tended to place less emphasis on direct seed extraction and more emphasis on the germination process. The germination method is more precise in this regard since it makes it possible to calculate the size of the actual weed seed bank by assuming that all viable seeds will eventually germinate, even if it takes many months of effort. The seedlings are also simpler to distinguish from the seeds.

Impact of Weed seed Bank Management on Crop Production

The management of weed seed banks is a first attempt to control weeds for future problems for sustainable crop production. Once weed seeds are managed in a weed seed bank, at least half the battle against weed management is won. The key to successful weed management is the long-term reduction of the weed-seed bank. This necessitates awareness of the seed bank's longevity, which can be influenced by weed populations, ecotypic variations among them, or the habitat of the plant that produced the seed. For instance, according to Gill and Fleet (2012) [57], barley grass (*Hordeum leporinum*) in a cropping system emerged later than barley grass on the field's roadside. The weed population in the field had evolved delayed emergence as a defense against pre-seeding and pre-emergent herbicides. According to Steinmann and Klingebiel (2004), annual ryegrass (*Lolium perenne*) planted in warm climates generated fewer, smaller, and less dormant seeds than plants grown in cool climates. Seeds produced by plants in cool, low moisture circumstances shed dormancy more quickly than seeds from plants receiving adequate water. Catherine and Abul (2020) [58] studying the emergence of weed seedlings over three years, reported that weed species with hard, woody seed coverings like Afghan melon, caltrop, double, and wireweed that have long-term seed banks lasted for at least three years while small-seeded species with broad seed dissemination had over 95% of their seeds emerge within the first year of seed production. Although the viability of the seed in the current research was not evaluated after three years, it is possible that some of the hard-seeded species would have persisted to emerge for another four to five years.

Evaluation of weed seed Bank

To acquire a thorough image of the weed seed reservoir in the soil profile, the determination of the soil weed seed bank is of utmost relevance. Hussain *et al*. (2017) [59] compared two methods i.e., sieve method and the seedling emergence method for determining the weed seeds in the weed seed bank. Soil samples from the experimental field were gathered from soil depths of 0–10 cm, 11–20 cm, and 21–30 cm before wheat was sown. They found that the sieve method had higher weed seed densities, weed frequencies, and more variety of weed species than the seedling emergence method, according to a comparative examination of seed bank extraction methods. As a result, it was decided that the sieve approach was better than the seedling emergence method. The sieving approach was found to be more accurate than the seedling emergence method in terms of cost, time, and user-friendliness, according to the feasibility analysis of seed bank extraction methods.

Weed seed Bank as a tool for Weed Management

The main focus of weed control is reducing weed seed rain and, by extension, weed seed bank. Because of this, controlling weed seed rain is the most crucial component of crop production and must therefore come first. For a thorough understanding of the weed seed bank, it is crucial to have a solid grasp of weed biology, including morphology, lifecycles, seed generation, etc. The hierarchy of weed management tools begins with controlling weed seed rain and the seed bank, then moves on to the potential and restrictions of rotations, soil quality, crop and pasture selection, pre-crop emergence weeding, particularly in stale seedbeds, crop establishment techniques, and finally, post-crop emergence weeding, which is regarded as the cherry on top of integrated weed management. Inverse to the deposit of money in a savings account, minimizing deposits and maximizing withdrawals are the basic objective in the direction of weed management. Hence, for effective weed management following efforts for minimizing deposits of weed seeds in soil should be practiced:

To be safe, kill weeds before they flower. Some weeds, like hairy Galinsoga, can mature seeds from flowers that are pollinated before the weeds are removed or cut. When in doubt, try to remove the seeds from blossoming weeds' fruits or blooms; mature seeds are those that have a dough-like consistency and are firm. If at all feasible, remove these seeds from the field. Before they may produce new rhizomes, tubers, or other propagules, eradicate creeping perennial weeds. Ensure that crops are grown before weeds since they may have less than 1% of the ability to produce seeds than robust weeds growing in full sun. Before they flower, walk through fields to eradicate huge weed escapes. Eliminating the top 10% of people can cut seed production by 90% or more. Field borders should be mowed to reduce weed species' capacity to enter fields through seed production. These borders also set a habitat for insect pests. After harvest, immediately mow or graze fields to stop weed seed generation. Use proper hygiene techniques to keep new weed species from entering the field, and get rid of new intruders quickly. The latter two deposits can introduce new weed species to the farm, similar to opening a new type of bank account with a little initial deposit and a high-interest rate. The first two deposits have the most influence on the population of future weed species. With just two or three viable seeds or propagules, a very aggressive new weed species could cause issues for years to come. Therefore, by keeping out their seeds and promptly eradicating new invaders, farmers seek to prevent both excessive deposits by spreading current weeds and the establishment of new weed species. Maximizing withdrawal of weed seeds from weed seed bank: aiming to stop weed seeds from flourishing in the soil can effectively withdraw weed seeds to remain in a weed seed bank. The following practices are suggested to achieve the maximum withdrawal of weed seeds from the weed seed bank:

Germination of seeds

The first kind of withdrawal—germination followed by emergence—is how weeds start to compete with and harm crops each season. It is also the primary method of debiting the seed bank, a useful tactic if newly emerged seedlings are quickly destroyed by cultivating the field by applying non-selective herbicides (the stale seedbed technique, for example). The majority of weed emergence from a particular season's seed rain occurs within two years of the seeds being shed, even in species with very long-lived seeds like pigweeds, velvetleaf, and morning glory [60]. Due to excessive depth or demise from allelochemicals (natural phytotoxic substances emitted by plants), microbial diseases, insects, or other organisms in the soil, fatal germination occurs when a seed or propagule grows but is unable to reach the soil surface.

Decay or Loss of Viability of Seeds over Time

Although the causes of aging are still largely unknown, it has been agreed that the process is unavoidable for a living entity. It is common knowledge that plant seeds lose viability when stored past the dormant stage. According to Van (2018) [61], data showed that the viability of seeds declines slowly from 98 to 80% during the first four years of storage but beyond that viability falls very quickly and reaches a significantly low value of 32% after 5 years. Nikolić *et al*. (2020) [62] conducting an experiment on Weed Seed Decay in No-Till Field and buffer strip reported that for all of the examined species, degradation of the buried seeds was greater in the no-till field soil than in the buffer strip, as was microbial cellulolytic activity. Although the no-till soil conditions were more unfavorable to seed viability, the buffer strip soil is an undisturbed ecosystem and had higher organic matter. They proposed that no-till management could enhance soil weed seed control. They observed that both seed degradation and viability % showed significant main effects and interactions ($p < 0.01$). For all of the examined species, degradation of the buried seeds was greater in the no-till (NT) field soil. *Digitaria sanguinalis* and *Alopecurus myosuroides* were the most severely deteriorated species in the no-till field, whereas *Abutilon theophrasti* and *Sorghum halepense* were the least severely degraded. The species in the buffer strip (BS) with the most deteriorated seeds was *D. sanguinalis*, which had 70% of its seeds degraded after 643 days of burial. In contrast, *S. halepense* had the least degraded seeds at the end of the experiment, with only 19% of its seeds degraded. They added that although *A. theophrasti*, a species with a thick and stiff seed coat, disintegrated quickly at the initial exhumation in both sites, the percentage of seed degradation remained stable at roughly 70% in the field and 50% in the BS during the subsequent exhumations. They opined that the physical characteristics of seeds appeared to be a crucial factor in determining the degree of degradation; species with thicker seed coats (*A. theophrasti, S. halepense*) were less deteriorated than those with thinner seed coats (*D. sanguinalis, A. myosuroides*). Naturally, weeds begin to compete with and destroy crops each season at the first type of withdrawal, which is germination followed by emergence. Additionally, it is the main way to debit the seed bank, which is a valuable strategy if newly sprouted plants are swiftly destroyed by additional care or fire (the stale seedbed technique, for example). Even in species with very long-lived seeds, such as pigweeds, velvetleaf, and morning glory, the majority of weed emergence from a specific season's seed rain happens within two years of the seeds being shed, according to Egley and Williams, (1990).

Changing Scenario of Weed Seed Bank in Soil

Once disseminated into a field, weed seeds can take on a variety of destinies. Some seeds thrive, emerge, and generate more seeds whereas others grow, die, decompose in the ground, or succumb to predators. Most weeds' seeds and other propagules have mechanisms that make some of them dormant (alive but unable to germinate) or conditionally dormant (will not germinate until given a certain stimulus, such as light), for variable amounts of time after they are shed. This aids the weed's ability to endure in an environment that is occasionally disturbed, hostile, and unpredictable. Weed seeds can transition from a dormant state to a non-dormant stage, where they can then germinate under a variety of environmental circumstances. Weed experts view dormancy as a means of dissemination through time since dormant weed seeds have the potential to produce future weed issues. Some seeds thrive, emerge, and generate more seeds whereas others grow, die, decompose in the ground, or succumb to predators.

Conclusion

Reducing the amount of weed seeds in the field and consequently limiting prospective weed populations during crop production is one of the most crucial, yet frequently overlooked weed management tactics. The weed seed bank has unlimited future and mechanism of weed seeds, many of which are poorly understood. Weed scientists have been prevented from fully comprehending the weed seed bank due to the sheer difficulties of monitoring a process that takes place primarily underground. Given the seed bank's inherent lifetime, different or extra weed control strategies may be necessary to cut down on the amount of weed seeds stored in the active seed bank. Aiming to reduce the number of weed species on a species-by-species basis while taking the geography and cropping system into consideration, management strategies must be created.

Application of research: The productivity of crops is influenced by weeds as they cause substantial reduction in yield. The persistence of weeds in soil is a major issue which survives for long time and germinates under favourable climatic condition. Hence, the foremost action for getting satisfactory crop yield, it is essential to understand the very nature of weed seed existing in weed seed bank and all efforts should be done to minimize their population and also suppressing their re generation to provide a weed free environment for crop production leading to higher yield

Research Category: Weed Management

Acknowledgement / Funding: Authors are thankful to Department of Agronomy, Birsa Agricultural University, Ranchi, 834006, Jharkhand, India

****Principal Investigator or Chairperson of research: Prof R. R. Upashni**
University: Birsa Agricultural University, Ranchi, 834006, Jharkhand, India
Research project name or number: Review study

Author Contributions: All authors equally contributed

Author statement: All authors read, reviewed, agreed and approved the final manuscript. Note-All authors agreed that- Written informed consent was obtained from all participants prior to publish / enrolment

Study area / Sample Collection: Department of Agronomy, Birsa Agricultural University, Ranchi, 834006, Jharkhand, India

Cultivar / Variety / Breed name: Nil

Conflict of Interest: None declared

Ethical approval: This article does not contain any studies with human participants or animals performed by any of the authors.
Ethical Committee Approval Number: Nil

References

[1] Buhler D.D., Hartzler R.G., Forcella F. (1997) *Weed Science*, 45, 329-336.
[2] Teo-Sherrell C.P.A., Mortensen D.A., Keaton M.E. (1996) *Journal of Applied Ecology*, 33, 1107-1113.
[3] Kellman M. (1978) *Journal of Biogeography*, 5, 291-300.
[4] Forcella F., Eradat-Oskoui K. and Wagner S. W. (1993) *Ecological Applications*, 3, 74-83.
[5] Singh A., Kaur R., Kang J.S., Singh G. (2012) *Agriculture and Health Sciences*, 1(1), 07-16.
[6] Cousens R., and Mortimer M.(1995) *Cambridge University Press*.
[7] Taylor K. L., and Hartzler R.G. (2000) *Weed Technology*, 14, 261-267.
[8] Blumenthal D., and Jordan N. (2001) *Weed Science*, 49, 509-519.
[9] Steinmann H. and Klingebiel L. (2004) *Weed Research*, 44 (5), 388-396.
[10] Thompson K., Grime J.P. (1979) *Journal of Ecology*, 67, 893-921.
[11] Walck J.L., Baskins J.M., Baskins C.C. (1996) *American Journal of Botany*, 83, 78-79.
[12] Thompson K., Ceriani R.M., Bakker J.P., Bekker R.M.(2003) *Seed Science Research*, 13, 97-100.
[13] Honda Y. (2008) *Plant Ecology*, 196, 301-309.
[14] Baskin C.C., Baskin J.M. (2006) *Weed Science*, (4), 549-557.
[15] Roberts H.A., Feast P.M. (1973) *Weed Research*, 13, 298-303.
[16] Lauren M. And Schwartz- Lazaro and Copes J.T.(2019) *Agronomy* , 9 (7),369.
[17] White S.S., Renner K.A., Menalled F.D. and Landis D.A. (2007) *Weed Science*, 55, 606-612.
[18] Cromer H.E., Murphy S.D. and Swanton C.J.(1999) *Weed Science*, 47, 184-194.
[19] Mauchiline A.L., Watson S.J., Brown V.K. and Froud Williams R.J. (2005) *Weed Research*, 45, 157-164
[20] Zhang J., Drummond A., Liebman M., and Hartke A. (1997) *Thesis, Maine Agricultural and Forest Experiment Station, University of Maine*.
[21] Westerman P.L., M. Liebman F. D. Menalled A. H., Heggenstaller, R. G. Hartzler, and P. M. Dixon. (2005) *Weed Science*, 53, 382-392.
[22] Shearin A. F., Reberg-Horton S. C. and Gallandt E. R. (2008) *Weed Science*, 56, 442-450.
[23] Chee-Sanford J.C., Williams II M.M., Davis A.S. and Sims G.K. (2006) *Weed Science*, 575-587.
[24] Jean-Luc Wolfender and Emerson Ferreira-Queiroz. (2018) *Press Release Geneva, August 28th*, 2018.
[25] Forcella F., Webster T., Cardina J. (2003) *In Weed Management for Developing Countries, Addendum; Labrada, R., Ed.; FAO: Rome, Italy*, 120, 3–18.
[26] Gardarin A., Guillemin J.P., Munier-Jolain N. M. and Colbach N. (2010) *European J. Agronomy*, 32, 162-168.
[27] Bouwmeester, H. J., and Karssen, C. M. (1992) Oecologia, 90, 88-94.
[28] Colbach N., Chauvel B., Durr C. and Richard G. (2002a) *Weed Research*, 42, 210-221.
[29] Colbach N., Dürr C., Chauvel B. and Richard G. (2002b) *Weed Research*, 42, 222-230.
[30] Badla D. and Benech A.R. L. (2005) New Phytology, 165, 445-452.
[31] Masin R., Zuin M. C., Archer D. W., Forcella F., and Zanin G. (2005) *Weed Science,* 53, 193-201.
[32] Benech-Arnold R. L., Sánchez R. A., Forcella F., Kruk B. C., and Ghersa C. M. (2000) *Field Crops Research*, 67, 105-122.

[33] Evans C. E., and Etherington J. R. (1990) *New Phytology*, 115, 539-548.

[34] Shrestha A., Roman E. S., Thomas A. G., and Swanton C. J. (1999) *Weed Science*, 47, 557-562.

[35] Grundy A. C., Phelps K., Reader R. J., and Burston S. (2000) *New Phytology*, 148, 433-444.

[36] Dorsainvil F., Dürr C., Justes E., and Carrera A. (2005) *European Journal of Agronomy*, 23, 146-158.

[37] Singh S. and Singh M. (2009) *Indian J. Weed Science*, 41(3 & 4), 134-145.

[38] Thill D., Schirman R., & Appleby A. (1979) *Weed Science*, 27(6), 625-630.

[39] Katsuyoshi N., Kanenori N. (1979) *Journal of weed Science and Technology*, 24(4), 233-239.

[40] Sharma R. K., Babu K. S., Chhokar R. S., Sharma A. K. (2004) *Crop Protection*, 23, 1049-1054.

[41] Clements D., Benoit D., Murphy S., & Swanton C. (1996) *Weed Science*, 44(2), 314-322.

[42] Buhler D. (1995) *Crop Science*, 35(5), 1247-1258.

[43] Hager A. (2013) *University of Illinois Extension. The Bulletin*. http://bulletin.ipm.illinois.edu/p385

[44] Cahoon C., Curran W. and Sandy D. (2018) *Grain Crops*, 103 -127.

[45] Ona Auškalnienė, Gražina Kadžienė, Daiva Janušauskaitė, Skaidrė Supronienė. (2018) *Zemdirbyste-Agriculture*, 105(3), 221–226.

[46] Koocheki A., Nassiri M., Alimoradi L .(2009) *Agronomy for Sustainable Development,* 29, 401-408.

[47] Buhler D.D., Kohler K.A., Thompson R.L. (2001) *Weed Technology*, 15, 170-176.

[48] Nichols V., English L., Carlson S., Gailans S. and Liebman M. (2020) *Front. Agron.,* 2, 591091.

[49] Burton N.R., Beckie H.J., Willenborg C.J., Shirtliffe S.J., Schoenau J.J., Johnson E.N. (2016) *Weed Science*, 64, 673-682.

[50] Walsh M., Newman P., Powles S. (2013) *Weed Technology*, 27, 431-436.

[51] Burnside O.C., Wilson, R.G., Weisberg S., Hubbard K.G. (1996) *Weed Science*, 44, 74-86.

[52] Toole E.H., Brown E. (1946) *Journal of Agricultural Researches*, 72, 201-210.

[53] Roberts H. A. and Neilson J. E. (1981) *Journal of Applied Ecology*, 18, 661-668.

[54] Burnside O.C., Moomaw R.S., Roeth F.W., Wicks G.A., Wilson R.G. (1986) *Weed Science*, 34, 248-251.

[55] Hussain M. M. and Begum M. (2015) *Journal of Bangladesh Agricultural University*, 13(2), 221-228,

[56] Grundy A.C., Mead A., Burston S. Modelling. (2003) *Journal of Applied Ecology*, 40, 757-770.

[57] Gill G.S. & Fleet B. (2012) *Weed Science,* 60, 394-400.

[58] Catherine B. and Abul H. (2020) *GRDC Research Development & Extension.*

[59] Hussain M., Ali S., Tahir M.N., Shah G.A., Ahmad I., Sarwar M.A. and Latif S. (2017) *Pakistan Journal of Agricultural Research*, 30(4), 310-315.

[60] Egley G. H., and Williams R. D. (1990) *Weed Science*, 38, 504-510.

[61] Van Treuren R., Bas N., Kodde J., Groot S.P.C., Kik C. (2018) *Conservation Physiology*, 25; 6(1), 33.

[62] Nikolic N., Squartini A., Concheri G., Stevanato P., Zanin G., Masin R. (2020) *Plants Basel*, 19(3), 293.