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## **ADVANCEMENTS IN DETERMINING THE ROLE OF BARLEY STRAW AS AN ALGAL CONTROL AGENT - NOTES**

### **Supporting Evidence**

- The first documented observation that algal growth might be affected by the presence of barley straw was made by Street (1977).
- Subsequent studies conducted within the United Kingdom have reported that barley straw can be used to control algae, under both laboratory (e.g., Gibson et al. 1990; Pillinger et al. 1994) and field conditions (e.g., Welch et al. 1990; Everall and Lees 1997). Upon decomposing in the water, barley straw was shown to inhibit the growth of both planktonic (e.g., Everall and Lees 1996) and filamentous algae, (e.g., Caffrey and Monahan 1999) as well as cyanobacteria, also known as “blue-green algae” (e.g., Barrett et al. 1996).
- Barley straw does not kill algae; it inhibits the growth of algae. Algae can recover and continue normal growth when placed in a non-treated environment (Gibson et al. 1990).
- The application of barley straw to reservoirs has been reported to lead to increased water clarity, reduction in the need to clean inflow and drainage filters, decreased odors (Barrett et al. 1999), and desirable aquatic plant reestablishments (Caffrey and Monahan 1999).
- Dosage levels that have led to desirable results have ranged from 5 to 50-g/m<sup>3</sup>.
- Martin and Ridge (1999) found that barley straw can inhibit both heterotrophic and photoheterotrophic growth of algae.
- Cooper et al. (1997) found barley straw to inhibit the growth of two species of *Saprolegnia* (water fungus).

### **Opposing Evidence**

- Norton et al. (1997) found ryegrass, oat chaff, lucerne, and red clover to show anti-algal activity, whereas barley straw showed no anti-algal activity.
- Martin and Ridge (1999) found barley straw to stimulate the growth of some algal species.
  - If an algal species is resistant to the straw, or if for some reason the straw does not become active, perhaps the added nutrients or carbon from the straw prompt this stimulation. Ball et al. (2001) found wheat straw to stimulate phytoplankton growth.

- Wills et al. (1999) attempted to use barley straw to control cyanobacteria (*Oscillatoria chalybea*) that was causing off-flavor problems in channel catfish; no consistent results were found.

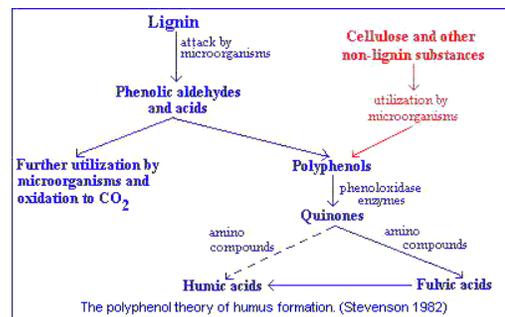
**Several hypotheses have been formulated; most have been disproved:**

1. The straw could be reducing the availability of some trace element or micronutrient essential for algal growth.
  - Welch et al. (1990) did not find any differences in concentrations of filterable reactive phosphorous, ammonium, or nitrate between a treated and non-treated section of a canal, yet, algal growth inhibition occurred in the treated section.
  - Gibson et al. (1990) added a substantial amount of N and P to an algal culture treated with barley liquor; algal growth inhibition still occurred.
2. The increased invertebrate populations associated with the straw (reported in Street 1997, 1978, and 1979) could be grazing on algae, consequently acting as a form of biological control.
  - This may be true to some extent; however, this cannot explain the significant reduction in algal growth documented by several controlled laboratory studies, e.g., Pillinger et al. (1994).
3. The effect could be due to pesticide residues on the straw.
  - Since barley straw takes weeks to months to become active, growth inhibition is not likely to be caused by pesticides, which would have an immediate impact, and would lose efficacy through time.
  - After analyzing water samples extracted from rotted barley straw that had been applied to a reservoir, Everall and Lees (1996) detected no suburea or triazine herbicides; significant algal growth inhibition occurred in this study.
4. Inhibitors produced by fungi attached to barley straw could cause the effect.
  - Pillinger et al. (1992) found three of several species of fungi to inhibit algal growth; however, they concluded that the general and widespread anti-algal effects of barley straw are unlikely to be explained by the anti-algal properties of specific fungi.
  - Ridge and Pillinger (1996) found autoclaved straw to still exhibit significant anti-algal activity. However, Gibson et al. (1990) found autoclaved straw to not become active, whereas non-autoclaved straw did.

5. Since barley straw did not become active in several studies until it had rotted, microbial decomposition could be essential; furthermore, the microbes that decompose straw could generate a naturally occurring algaecidal chemical.
- Ball et al. (2001) found that an extract prepared from finely chopped fresh barley straw did not inhibit the growth of *Microcystis*, whereas an extract prepared from finely chopped decomposed barley straw did.
  - Pillinger et al. (1994) and Ridge and Pillinger (1996) concluded that since finely chopped, fresh straw also exhibited anti-algal effect, microbial decomposition is not essential; hence, it is the straw that produces the effect, not the microbes.
    - In the field, the microbes are most likely needed to break down the straw, as the latter researchers did mechanically.
6. Once broken down, the straw releases an anti-algal chemical, or a chemical that, through a pathway, becomes anti-algal.

- Pillinger et al. (1994) suggested that phenolics present (tannins) in the straw could be responsible for the anti-algal effect of barley straw; barley straw does contain tannins (Brandon et al. 1982).
  - They found an authentic source of tannin, tannic acid, as well as a natural source, oak leaves, to be anti-algal.
  - When hide powder, a protein source expected to complex with tannin, was added to the tannic acid and to the oak leaf extract, the inhibitory effect of both was diminished. However, this only occurred at acidic pH values; the protein precipitation did not occur with tannic acid or the natural tannin source (oak leaves) under alkaline conditions (pH~8).
  - Quinones (oxidized phenolic compounds) were also tested. The study showed strong evidence that oxidized phenols are much more efficacious.
  - Increasing aeration during the decomposition of barley straw and during bioassays of rotted straw liquor and fresh, finely chopped barley straw significantly increased growth inhibition.
- The latter authors stated that further oxidation of quinones would lead to the formation of humic acids, which perhaps would nullify the algal growth inhibitory effect. This thought fits two patterns.

First, studies (e.g., Harriman et al. 1997) have reported that the effect of barley straw diminishes in about six months; perhaps by this time humic substances have formed. Secondly, Kholdegarin and Oertil (1997) reported phenolic compounds to interfere with nitrification, and Vallini et al. (1997) reported humic acids to facilitate nitrification.



- However, recent unpublished reports as well as some published reports (e.g., Barrett et al. 1996) have speculated that these humic substances, in the presence of sunlight, create conditions suitable for the formation of hydrogen peroxide, and that this is the substance responsible for the algal growth inhibition.

- Hydrogen peroxide is formed in natural waters through the interaction of humic substances and sunlight (Cooper et al. 1988). Hydrogen peroxide has been shown to inhibit the growth of algae (Barrett and Newman 1992). Hydrogen peroxide inhibits carbon dioxide fixation (Ashton and Crafts 1981).
- However, Pillinger (Irene) found barley straw to inhibit the growth of algae in the dark, which is obviously a condition that lacks sunlight to induce hydrogen peroxide formation.
- Pillinger et al. (1995) tested another phenolic source (lignin) taken from rotted elm and sycamore wood for anti-algal properties; barley straw contains lignin (Love et al. 1998; Ball et al. 2001).
    - Both white-rotted and brown-rotted wood of both species showed growth inhibition ability.
    - Brown-rotted wood was found to be much more inhibitory; this was attributed to its higher lignin content.
    - This finding was supported in Ridge and Pillinger (1996).
  - Everall and Lees (1997) analyzed water samples extracted from rotted barley straw that had been applied to a reservoir; significant algal growth inhibition occurred in this study
    - The authors attributed the effects to a variety of phenolic compounds found in the samples. One of the compounds analyzed, 2,6-dimethoxy-4-pheno, was found to be highly inhibitory to algal growth in Pillinger (1994).
  - Ridge and Pillinger (1996) tentatively concluded that algal inhibition by barley straw is associated with the solubilization and oxidation of lignin, and furthermore, that the source of algal inhibition can be more accurately defined as oxidized polyphenolics.

### **Management Considerations**

- Chemical algaecides such as copper sulfate are not used (from a practical standpoint) until heavy algal coverage has occurred, whereas barley straw potentially can be used as a preventative measure because of its growth inhibitory effect.
  - Implications:
    1. Large die-offs of algae caused by treatment with an algaecide can create dissolved oxygen voids, increases in toxic ammonia, and pH fluctuations.
      - o If applied early enough in the season, barley straw may hinder the initial blooms of algae, and suppress growth for approximately six months, consequently eliminating the above effects.
    2. Because algae can quickly reestablish in nutrient-rich waters after being killed with an algaecide, multiple treatments are needed each summer. This may involve great expense and labor.
      - o Regardless of the concentrations of growth-mediating nutrients in the water, algae may not reestablish for six months after applying a

single treatment of barley straw. If new straw is added before the old straw has lost efficacy, this growth suppression may be continued indefinitely.

- Caffrey and Monahan (1999) found that stands of *Myriophyllum* (water milfoil) had reestablished in a barley-treated section of a canal, after significant algal reduction had occurred. To the best of my knowledge, there has been no reporting of barley straw inhibiting the growth of vascular plants.
- Although numbers of algal cells were reduced, Harriman et al. (1997) and Barrett et al. (1999) reported that diversity was not affected. If barley straw's effect was selective in these studies, such that it did not target all of the species, then the unaffected species may have proliferated due to the lack of competition; this finding is a definite plus when considering barley straw for management purposes.
- If substantial algal coverage has already occurred, both a chemical algaecide and barley straw could be used in conjunction. Since barley straw cannot kill algae already present, its inhibitory effect may not be noticed in this instance, unless for some reason the algae already present die. While the straw is rotting, a chemical algaecide could be used to clear the pond of algae, at which time the barley straw may inhibit the algae's recovery.
- Barley straw should be applied either during early spring at least one month ahead of the first bloom, or during late winter, in which case the straw can be placed on top of the ice-covered pond. However, the latter method should be avoided if a pond is suspected to suffer from dissolved oxygen voids over the winter season.
- The straw should be wrapped fairly loosely in some form of mesh net to allow the water to circulate through the straw.
- Barley straw should be placed in an oxygen-rich environment, such as near the surface in deep water, or near an inflow. Although the shallow edges of ponds are usually high in dissolved oxygen during the day from the intense photosynthesis of vegetation in these areas, these areas may become very low in dissolved oxygen during the night due to the respiration of the vegetation. In addition, dense populations of vegetation in these areas may also impede with the movement and interstitial-exchange of the water, again consequently reducing oxygen, and perhaps also hindering the movement of straw's leachate. Placing the straw near the surface may aid in the distribution of the chemical from the wave action of the surface water. Oxygen may aid in the efficacy of the straw for two reasons:
  1. Oxygen is necessary for microbes and other organisms to decompose the straw.
  2. Oxidized phenolic compounds may be more efficacious.
  3. Oxygen is required for the production of hydrogen peroxide in water.

- Aerators in ponds would be very advantageous, as they would increase oxygen levels and spread the straw's leachate throughout the pond. The aerator should be placed in the center of the pond, and some of the barley bales should be placed around the aerator.
- If the straw will be positioned in deep waters, an anchor and float should be used to keep the straw submerged, yet near the surface.
- Avoid placing the straw near drains.
- Avoid placing barley straw in areas known to become infested by higher plants such as sago pondweed; infestations may reduce available oxygen via competition and reduced water movement, and prevent the straw's leachate from spreading throughout the pond.
- CAPN (1995) reported that concentrations might need to be increased when applying the straw to turbid or muddy waters.
- Most of the published literature reported that barley straw required at least one month to become effective. Most of the unpublished information reports that barley straw can become effective in two weeks at warm temperatures.
- All of the published literature mentioned here has reported concentrations as  $\text{g/m}^3$ . However, most of the unpublished literature has reported the required concentrations as  $\text{g/m}^2$ . Although using area is easier, it should be remembered that the results shown were based on volume.
- I recommend using 2-3 bales (1 bale ~ 40 lbs) barley straw per acre depending upon availability and feasibility.

**Table 1. Estimation of barley straw required to obtain various concentrations.**

Level/ Concentration	Measurement Unit	Assumed Average Depth (Feet)	Pounds of Barley Straw Per Acre	# bales (40lbs each)
5	$\text{g/m}^3$	5	68	1.7
10	$\text{g/m}^3$	5	136	3.5
15	$\text{g/m}^3$	5	204	5.1
30	$\text{g/m}^3$	5	408	10.2
5	$\text{g/m}^3$	10	136	3.5
10	$\text{g/m}^3$	10	204	5.1
15	$\text{g/m}^3$	10	408	10.2
30	$\text{g/m}^3$	10	815	20.3
5	$\text{g/m}^2$	---	45	1.1
10	$\text{g/m}^2$	---	89	2.2
15	$\text{g/m}^2$	---	134	3.4
30	$\text{g/m}^2$	---	267	6.7

**I. Sample Calculation For Volume (  $\text{g/m}^3$  ):**

1 acre =  $43560 \text{ ft}^2 \times 5 \text{ ft}$  (average depth) =  $217800 \text{ ft}^3 = 6167 \text{ m}^3$ ;

$$\frac{5 \text{ g}}{\text{m}^3} \times \frac{6167 \text{ m}^3}{1 \text{ acre}} = \frac{30835 \text{ g}}{\text{acre}} \times \frac{1 \text{ lb}}{454 \text{ g}} = \frac{68 \text{ lbs}}{\text{acre}}$$

**II. Sample Calculation for Area (  $\text{g/m}^2$  ):**

1 acre =  $43560 \text{ ft}^2 = 4047 \text{ m}^2$ ;

$$\frac{5 \text{ g}}{\text{m}^2} \times \frac{4047 \text{ m}^2}{\text{acre}} = \frac{20235 \text{ g}}{\text{acre}} \times \frac{1 \text{ lb}}{454 \text{ g}} = \frac{45 \text{ lbs}}{\text{acre}}$$