

Evaluation of several end-of-pipe measures proposed to control cyanobacteria

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Abstract While reduction in nutrient loading is a prerequisite for mitigation of harmful cyanobacterial blooms in nutrient-enriched waters, in certain surface waters eutrophication control is not always feasible due to practical and economic constraints or might be effective only in the long run. Yet, the urgent need to control cyanobacteria in water for drinking, irrigation, aquaculture, industry and recreation has spurred the development of a plethora of alternative methods that claim to be fast acting. Here, we provide a critical overview of several of these end-of-pipe measures: effective microorganisms (EM[®]), golden algae (*Ochromonas*), plant/tree extracts, ultrasound and artificial mixing of non-stratifying waters. Most of the end-of the pipe measures claim to provide

sustainable control of harmful cyanobacterial blooms, while at best only targeting symptom relief rather than eutrophication relief. Support for “effective” microorganisms, golden algae, plant extracts, ultrasound and artificial mixing of non-stratifying waters to diminish eutrophication problems such that the resulting water quality meets societal and legislation demands is limited, and several proposed underlying mechanisms are doubtful. None of these curative measures seem the desired wide applicable solution to cyanobacterial nuisance; they should not be considered Columbus’s egg. A critical evaluation of end-of pipe measures is crucial for water authorities in their choice for mitigating measures.

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Introduction

Excessive nutrient loading is the major cause of water blooms of cyanobacteria (blue-green algae), i.e., elevated densities throughout the water column (Conley et al. 2009; Smith et al. 1999). Water blooms may cause high turbidity and mal odor of the water, while associated nocturnal oxygen deficiency can lead to fish kills (Paerl and Huisman 2008; Smith et al. 1999). Such blooms may have significant detrimental

environmental impacts by food web changes reducing biodiversity (Paerl 2008; Paerl et al. 2001). Cyanobacterial blooms have caused drinking water shortages (Yang and Liu 2010) and pose a serious health threat because cyanobacteria might produce potent toxins (Codd et al. 2005; Dittmann and Wiegand 2006). This is especially the case when in a stable water column in lakes and ponds floating layers or surface scums develop that might be further concentrated on the leeside shore, resulting in a manifold concentration of the mostly intracellular contained toxins (Chorus et al. 2000).

Because of the paramount role of nutrient loading in the development of cyanobacteria blooms and scums, nutrient input reductions are the most obvious targets in controlling cyanobacteria harmful blooms (Paerl and Otten 2013). The reduction in external nutrient loading is a prerequisite for improvement, but lake recovery can be delayed for decades due to internal phosphorus loading (Søndergaard et al. 1999). Consequently, hazardous cyanobacterial blooms may remain in these waters for many years. In other systems, such as open systems with intense agricultural influence, nutrient input reductions may not always be possible and cyanobacteria blooms will sustain. Such blooms are clashing with modern society's demand for good water quality (Steffensen 2008) and are in conflict with the attainment of a good water quality needed to comply with both the EU Water Framework Directive (WFD, European Union 2000) and the EU Bathing Water Directive (BWD; European Union 2006). Despite water authorities displaying a great need for preventive measures leading to nutrient reduction and thus to the reduction in cyanobacteria blooms in the long term, they also need short-term fast acting, curative treatments to mitigate cyanobacterial nuisance.

Curative methods should rapidly suppress the proliferation of cyanobacteria or destroy a massive bloom bringing immediate improvement of the water quality and should provide, at least in the growing season, access to the water for drinking, irrigation, aquaculture, industry and recreation (Jancula and Maršálek 2011). Several of these curative measures, such as the application of cyanocides and algicides (Jancula et al. 2016), and manipulations of the food web through macrophytes (Bakker and Hilt 2016) and artificial mixers (Visser et al. 2016) have been dealt with elsewhere in this special issue. Nonetheless,

many products remain of which quite a number often have been proposed to water authorities as end-all solutions in controlling cyanobacteria, especially following upon the typical heat waves of 2003 and 2006 with numerous cyanobacterial bloom events in northwestern and central Europe. Here, we will critically review the claims and effectiveness of a selection of methods: i.e., “effective” microorganisms, golden algae, plant extracts and ultrasound. The choice for this selection is based on the scientific information available and the strong promotion or media attention the products received in the Netherlands over the last decade, where golden algae got quite some media attention, questions about “effective” microorganisms made it into the National Parliament, while ultrasound, Barley straw and SolarBee[®] trials were conducted in situ. A critical review of these end-of pipe measures might be helpful to water authorities in making a more balanced decision for effective treatments to control eutrophication and mitigate cyanobacterial nuisance.

Effective microorganisms (EM[®])

The use of effective microorganisms (EM) has been advocated to be an end-all solution to a wide suite of water quality problems (e.g., URL1–5; Zakaria et al. 2010). A blend of EM can be kneaded into dried mud to form mud balls that can be thrown in water bodies. These “EM-mudballs” are based on a concept that was first developed by Higa (1998), who suggested that—through competitive exclusion—addition of EM-1[®] changes the microbial community toward dominance of beneficial species, while suppressing harmful bacteria. EM cocktails are allegedly said to contain about 80 species of microorganisms, such as photosynthetic bacteria, lactic acid bacteria, actinomycetes, yeasts and fermenting fungi (Higa 1998). However, this could not be confirmed by analysis of EM-1[®] samples that revealed the majority consisted of lactic acid bacteria (*Lactobacillus* and *Lactococcus* at $5\text{--}10 \times 10^6 \text{ mL}^{-1}$) and yeasts (*Saccharomyces* and *Candida* at about 10^5 mL^{-1}), while other microorganisms were present in very low concentrations or not present at all (Van Egeraat 1998). Semiquantitative PCR-DGGE could not confirm the stated richness of EM either (Van Vliet et al. 2006).

EM claims range from water purification to sustainable end-all solution for water quality and sanitation problems. These claims can be found on various webpages and are based on anecdotal evidence rather than on scientific confirmation with reproducible and consistent data (Higa and Parr 1994). For example, the statement “Using EM-1[®] Microbial Inoculant on a regular basis will help to keep enough beneficial microbes in the system to keep nutrient levels low enough to prevent the growth of algae” (URL6) is not supported by any scientific study.

There are only limited scientific studies on the use of EM to control nutrients or cyanobacteria in aquatic systems. A recent study showed that the combination of EM and submerged plants, *Hydrilla verticillata*, had good removal of total nitrogen (TN) and total phosphorus (TP) (Chen et al. 2013). Also humic substance removal by EM was studied, and although the authors report good removal of humic substances by EM (Joo and Foldenyi 2012), the lack of proper controls should be noted. In a 1200-m² pond in Poland fed by purified water from a sewage plant, several water quality variables were measured in a period before and after the addition of EM (Jóźwiakowski et al. 2009). Although the preliminary data of Jóźwiakowski et al. (2009) indicate a decrease in both TN and TP, the limited sampling regime (twice in winter, once in spring) does not allow for drawing definite conclusions on the effectivity on the application of EM. Application of four mudballs and 2.5 L liquid material both containing EM to a 24-m² garden pond in Hungary led to elevated SRP levels and lower transparency indicating the inefficacy of the EM in controlling eutrophication (Padisák 2014).

Controlled experiments in the laboratory found no growth inhibition for a laboratory strain of *Microcystis aeruginosa* and for *M. aeruginosa* from the field at a recommended dosage of 1 EM-mudball per square meter ($\approx 0.1\text{--}0.3\text{ g L}^{-1}$) (Lürling et al. 2009; 2010). Suspensions of EM-mudballs up to 1 g L^{-1} were ineffective in reducing cyanobacterial growth. Cyanobacteria were inhibited only at very high EM-mudball concentrations ($5\text{--}10\text{ g L}^{-1}$), because of the very high amount of clay and high water turbidity (Lürling et al. 2009; 2010). Also a suspension of “activated effective microorganism” (EM-A) was not effective in reducing cyanobacteria growth and high concentrations of EM-A caused low pH of the water and nutrient enrichment (Lürling et al. 2009). High

dosage of EM-mudballs caused water column oxygen depletion (Lürling et al. 2009, 2010), release of metals (Al, Cd, Cu, La and Pb) and release of phosphate (Lürling et al. 2009). Hence, such “effective microorganisms” formulations are not effective to prevent the growth of algae or in preventing cyanobacterial proliferation and/or terminating blooms. The Dutch authorities prohibited the application of EM-mudballs in Almere-Haven, because they contained heavy metals such as mercury as well as nutrients (Rijkswaterstaat 2007).

Overall, the claim that EM will “keep nutrient levels low enough to prevent the growth of algae” in surface water is not supported by scientific evidence. In contrast, controlled experiments have clearly shown that cyanobacteria cannot be controlled with EM.

Golden algae (*Ochromonas*)

In August 2009, a press release based on a publication of a study with *Microcystis* and the chrysomonad *Ochromonas* (Van Donk et al. 2009) led to more than a dozen news items in the Dutch media with main emphasis on the green nature of this promising solution, and a comparison with the use of natural enemies such as predatory mites in horticulture pest control was made (source: LexisNexis[®] Academic). Consequently, several water authorities considered the use of the shiny golden alga *Ochromonas* in controlling cyanobacteria.

It is already known for decades that chrysomonads (*Ochromonas danica*, *O. minuta* and *Poteroochromonas malhamensis*) can feed on cyanobacteria (Cole and Wynne 1974; Daley et al. 1973). They can ingest food items several times larger in diameter than their own which is around $10\text{ }\mu\text{m}$ (Zhang et al. 1996), seemed to modify or degrade microcystins (Ou et al. 2005) and are capable of growing on sole cyanobacterial food (Zhang and Watanabe 2001). Chrysomonads, however, had no significant effect when *Microcystis* density was high (Zhang et al. 2009a). On the other hand, *Microcystis* can respond to the threat of being grazed by chrysomonads by building colonies (Burkert et al. 2001; Wang et al. 2010). Formation of large colonies in *Microcystis aeruginosa* was observed under continuous grazing pressure by *Ochromonas* that allowed *Microcystis* population growth (Yang and Kong 2012). Those colonies were protected from *Ochromonas* grazing, which was further

corroborated in studies that showed low clearance rates on colonial *M. aeruginosa* compared to unicells (Yang et al. 2009b, c). Thus, a common bloom-forming organism like *Microcystis* had simply grown to sizes beyond the ingestion capacity of the chrysomonads. In eutrophic systems, the summer increase in phytoplankton contains a high proportion of inedible species (Sommer et al. 2012). There is no evidence that chrysomonads would be capable of grazing down those grazing-resistant phytoplankton, often comprised of large dinoflagellates and/or cyanobacteria (Sommer et al. 1986).

The chrysomonads themselves are on the menu of several larger metazoan grazers, mesozooplankton (Saunders et al. 1994). They can be toxic to mesozooplankton (Boxhorn et al. 1998; Leeper and Porter 1995; Hiltunen et al. 2012), but it is assumed that at normal field abundances of these organisms toxic effects will be hardly detectable (Boenigk and Stadler 2004). Grazing by chrysomonads on *Microcystis* can also reduce the toxicity of *Microcystis* to *Daphnia* (Zhang et al. 2009b). Therefore, chrysomonads are often controlled by mesozooplankton (Sommer et al. 2012).

While *Ochromonas* and *Poterioochromonas* might be widespread and common mixotrophs (Boenigk and Stadler 2004; Van Donk et al. 2009), the incidence and intensity of cyanobacterial blooms have increased over the last decades (O'Neil et al. 2012), strongly suggesting that the chrysomonads fail to control cyanobacteria in the field. Mass culture and depositing chrysomonads in the field are advised against, because of the expected lack of effect and the addition of nutrients to the receiving water.

Plant/tree extracts

A whole array of plant extracts and chemicals exuded by macrophytes for controlling cyanobacteria is promoted. In a thorough review, it was concluded that “With many findings on new allelochemicals, it will become the most promising method to control algal bloom” (Hu and Hong 2008). Among the most studied is barley straw (extracts) that could reduce growth of unicellular and filamentous green algae (Welch et al. 1990; Gibson et al. 1990) and cyanobacteria such as *Microcystis aeruginosa* (Newman and Barrett 1993). During straw decomposition under UV-supplemented visible light, hydrogen peroxide was

produced that might have caused inhibition of *M. aeruginosa* (Iredale et al. 2012). This could explain the differences in sensitivity between eukaryotic algae and cyanobacteria as cyanobacteria in general are more sensitive to hydrogen peroxide than green algae and diatoms (Drábková et al. 2007). The active compounds, polyphenols (Pillinger et al. 1994; Ridge and Pillinger 1996), have recently been identified as salcolin A and B (Xiao et al. 2014). Salcolin A caused after 1 day an increase in *M. aeruginosa* intracellular reactive oxygen species (Xiao et al. 2014). The findings have triggered speculations on the use of barley straw for controlling *M. aeruginosa* blooms (Shao et al. 2013) and more specific the use of salcolin in the “future control of cyanobacterial harmful algae blooms” (Xiao et al. 2014). However, a protective role of microcystins against oxidative stress has been suggested too (e.g., Phelan and Downing 2011), which finds further support in an experiment where a microcystin producing *M. aeruginosa* strain was better protected against oxidative stress caused by hydrogen peroxide than its microcystin-free mutant (Zilliges et al. 2011). Hence, increased oxidative stress caused by the decaying barley straw could potentially select for more toxic (microcystin producing) cyanobacteria.

Applying rotting barley straw (50 and 25 g m⁻³) to shallow reservoirs inhibited phytoplankton and lowered cyanobacteria dominance (Everall and Lees 1996; 1997). Repeated addition of barley straw (6–28 g m⁻³) to a reservoir lowered phytoplankton and cyanobacteria cell numbers to one half or to one quarter (Barrett et al. 1999). A pond experiment with different barley straw dose (40 and 80 g m⁻²) revealed inhibition of *M. aeruginosa*, *Anabaena* sp. and *Aphanizomenon* sp., no effect on *Nostoc* sp. and a stimulation of *Oscillatoria* sp. (Rajabi et al. 2010), while another pond experiment showed no difference between controls and barley-treated ponds (Ferrier et al. 2005). The phytoplankton abundance in a loch treated with barley bales declined after 10 months compared to an untreated loch and remained low for subsequent 17 months (Harriman et al. 1997). In contrast, in another Scottish loch, barley had no effect on phytoplankton and no evidence for such a delayed effect of straw bale placement was detected as April placement was followed by an August bloom (Kelly and Smith 1996). Likewise, adding barley straw into a rice field did not reduce phytoplankton abundance (Spencer and Lembi 2007) and in an enclosure study

three different barley concentrations (5, 15 and 60 g m⁻³) showed no effects on algae (Boylan and Morris 2003). The latter authors mentioned a case of two ponds where a combination of barley straw and aerators reduced phytoplankton biomass (Boylan and Morris 2003). Positive effects of barley straw on controlling *Microcystis* and cyanobacteria for some reservoirs in the UK were reported (Purcell et al. 2013), but these find no strong support in the underlying data.

In 1997 and 2000, barley straw was added to an urban pond in Roosendaal (the Netherlands) and in 2000 to a small lake near Waspik. For the latter, mean summer chlorophyll-*a* concentrations were 156 µg L⁻¹ in 1999 prior to barley and 106 µg L⁻¹ in 2000 after the barley placement (Water Authority Brabantse Delta). In the urban pond Parkvijver (Roosendaal, the Netherlands), mean summer chlorophyll-*a* concentrations were 167 µg L⁻¹ in 1995 before barley placement, only 23 µg L⁻¹ in 1997 during barley presence, 30 µg L⁻¹ in 1998, 116 µg L⁻¹ in 1999, while after a second barley placement in 2000, mean summer chlorophyll-*a* concentrations were 306 µg L⁻¹. In 1999, phytoplankton was dominated by chlorophytes reaching a maximum phytoplankton cell concentration of 3.4 × 10⁵ cells mL⁻¹ in August, while less than 3 months after the barley placement (beginning March 2000), cyanobacteria already flourished reaching densities of 7.7 × 10⁵ cells mL⁻¹ on May 29, 2000 (Fig. 1). End of July, chlorophyll-*a* and phytoplankton concentrations had dropped, but they increased again after a second application of barley straw in the beginning of August 2000 (Fig. 1).

The mixed results that emerge from field trials have also been obtained in more controlled laboratory assays. In general, *M. aeruginosa* appears among the most vulnerable organisms (Martin and Ridge 1999; Ferrier et al. 2005). However, not all cyanobacteria seem susceptible to rotting barley straw; no effects were observed in *Pseudoanabaena* sp. (Brownlee et al. 2003) and growth stimulation has been observed in *Anabaena cylindrica*, *A. flos-aquae*, *Oscillatoria animalis* and *O. lutea* var. *contorta* (Martin and Ridge 1999; Ferrier et al. 2005). Similarly, barley straw extract had variable effects on dinoflagellates, inhibition of some *Heterocapsa* species, no effect or stimulation of Gymnodiniales and stimulation of Prorocentrales (Terlizzi et al. 2002). Commercially available barley straw extract had no effect on the

ichthyotoxic *Prymnesium parvum* (Grover et al. 2007). Likewise, testing microbe-lift barley straw concentrated extract (1.5 × 10⁻⁴ to 15 mL L⁻¹) in our laboratory on *M. aeruginosa* (initial chlorophyll-*a* concentration 17 µg L⁻¹) revealed no growth inhibition (Fig. 2).

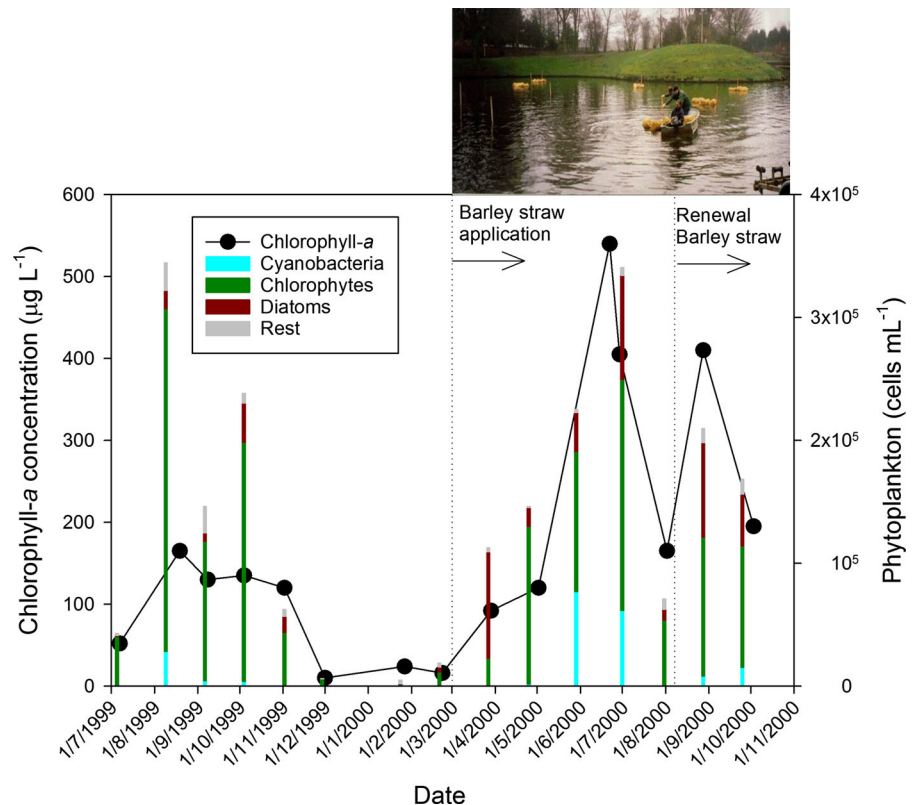
Decomposed barley straw extract inhibited *M. aeruginosa*, while fresh extract promoted growth (Ball et al. 2001). Microbial activity appears essential for barley straw to become inhibitory causing a lag period before straw becomes “active.” Fine chopping of the straw can shorten this lag phase substantially (Iredale et al. 2012).

As with effects on cyanobacteria, also varying effects on water quality variables have been reported. For example, while in an enclosure study barley straw (1.3 g L⁻¹) was effective in suppressing *Aphanizomenon flos-aquae*, dissolved oxygen levels dropped to near zero (Haggard et al. 2013). In contrast, others found higher oxygen levels in barley-treated ponds (Rajabi et al. 2010) or no effects in an enclosure study (Boylan and Morris 2003). Moreover, the addition of barley straw to mesocosms increased phosphate levels fivefold to tenfold compared to the control (Haggard et al. 2013), whereas in another study, no effects on nutrients were found (Boylan and Morris 2003).

Although widely accepted that barley straw can be an “effective control method” (Purcell et al. 2013), an “effective and environmentally-sound option for the control of cyanobacterial and microalgal blooms” (Iredale et al. 2012) and “very useful for controlling of *M. aeruginosa* based blooms” (Shao et al. 2013), the lag phase, the different sensitivity of organisms, the contrasting field results and the proposed underlying mechanism of oxidative stress imply that use of rotting barley straw should be met with care.

In addition to barley straw extract, many plant-derived chemicals have anti-cyanobacterial activities. These compounds are mostly extracted from plant tissue and then tested on cyanobacteria, mainly *M. aeruginosa* (Table 1). Controlled experiments with extracts of *Fructus mume*, *Salvia miltiorrhiza* and *Moringa oleifera* as well as L-lysine and D-lysine gave no support that these plant extracts and amino acid could be promising candidates for curative application in *M. aeruginosa* bloom control (Lürding and van Oosterhout 2014). From several studies, it is unclear whether the effect is caused by the extract or by the solvent. Often rather high concentrations are needed,

Fig. 1 Phytoplankton abundance (bars) for cyanobacteria (blue fill), chlorophytes (green fill) and diatoms (brown fill) as well as the course of the chlorophyll-*a* concentration (solid black line) over the period July 1999–September 2000. The two vertical dashed lines represent the addition of barley straw. A photograph of the application is added. (Data from: AquaSense 2000; Bijkerk 2000)



making applications expensive or virtually impossible (Shao et al. 2013). Moreover, concomitant input of nutrients could aggravate eutrophication (Lüring and Beekman 2010; Shao et al. 2013), while high amounts of organic matter in combination with expected warming could lead to higher bloom toxicity (Ekvall et al. 2013).

Plant extracts are often used as synonyms for allelochemicals, despite no proof on their exudation from living plants to the surrounding water exists. The effects of living plants on mitigation of cyanobacterial nuisance are reviewed elsewhere in this issue (Bakker and Hilt 2016). Here we add a few words on allelochemicals, as they might contribute to stabilization of clear water states in shallow lakes (Hilt and Gross 2008). Probably, the life time of active compounds is rather short. For example, in a study with spent medium of *Stratiotes aloides* in which two strains of *M. aeruginosa* were grown, a delayed lag phase was observed, but as exponential growth rates were equal or slightly higher than the controls (Mulderij et al. 2005), these exposed cyanobacteria apparently caught up rapidly pointing toward rapid decline of the active

compounds. Hence, it remains to be seen whether field applications of plant extracts or allelochemicals can produce a window of clear water long enough for submerged and presumably allelopathic active macrophytes to establish. Furthermore, a recent study showed while *M. aeruginosa* was inhibited by macrophyte allelochemicals when growing in pure culture, interacting with a green alga completely reversed inhibition into enhancement (Chang et al. 2012). This led the authors to conclude “allelopathically-active macrophytes might thus support cyanobacteria rather than suppress them in situ” (Chang et al. 2012).

Ultrasound

Ultrasound is sound of frequencies higher than those that can be detected by the human ear (Mason 2007), i.e., approximately 20 kHz and higher. The frequency range ~20–200 kHz is considered low-frequency ultrasound and used in industry and therapy, while frequencies up to 20 MHz are applied in medical diagnostics (Ahmadi et al. 2012). Ultrasound finds a

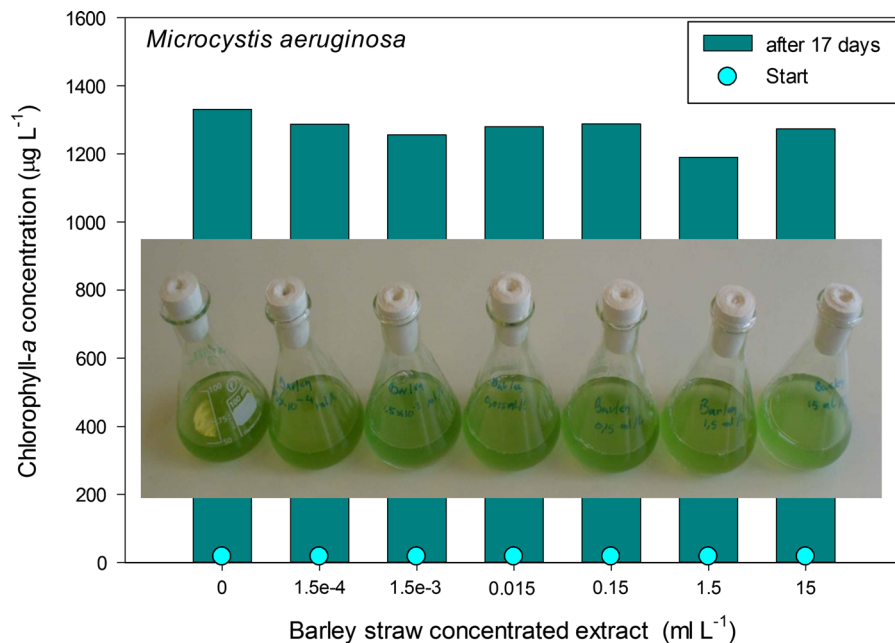


Fig. 2 Chlorophyll-*a* concentrations of *Microcystis aeruginosa* after 17-day growth in different concentrations of microbe-lift barley-concentrated extract (0–15 ml L⁻¹). *M. aeruginosa* was grown in 100-mL Erlenmeyer flasks containing 50 mL of autoclaved WC medium (Lürling and Beekman 2006) that were placed in a Gallenkamp ORBI-SAFE Netwise Orbital Incubator

at 20 °C, in 60 rpm and in a 18:6-h light:dark rhythm that was programmed to gradually increase light intensity to a maximum of 130 µmol quanta m⁻² s⁻¹ and subsequently decreased again to darkness, which resulted in a daily average light intensity of ~57 µmol photons m⁻² s⁻¹. A picture of the cultures after 17-day incubation is inserted

large range of applications in medicine, science, industry, including various water treatments (Phull et al. 1997). The alleged potential of ultrasound controlling cyanobacteria in situ is based on several laboratory studies showing clear effects of ultrasound on cyanobacterial growth, the collapse of gas vesicles, cell wall disruption and disturbance of the photosynthetic activity (Wu et al. 2011; Rajasekhar et al. 2012a). The vast majority of these laboratory studies have used high-power devices (e.g., 20–80 W, Hao et al. 2004a; 40–1200 W, Lee et al. 2001) that cause acoustic cavitation: a process in which compression and rarefaction create gas bubbles that may collapse (Neppiras 1980). On collapse of the bubbles, several processes such as pressure gradients, shear forces, formation of radicals and hydrogen peroxide production may disrupt the cells (Joyce et al. 2003). However, such relatively high ultrasound intensities are difficult to apply in lakes and ponds as in larger volumes significantly less power is transmitted, and consequently, the impact on cyanobacteria will be far less (Rajasekhar et al. 2012a). This finds support in

some field studies using higher power units—10 units of 2 times 100 W (200 kHz) in a 365.000-m³ reservoir (Lee et al. 2002) and one 630-W unit (22 kHz) in 9000-m³ pond (Ahn et al. 2007). The pond study of Ahn et al. (2007) gave no support for strong cyanobacteria control by ultrasound, because both the control and treated pond were dominated by diatoms and green algae, the treated pond already at start had significantly lower chlorophyll-*a* concentration than the control, while at the end of the experiment, chlorophyll-*a* concentrations in both ponds were the same. Lee et al. (2002) reported that chlorophyll-*a* concentrations were lower in the 2 years of ultrasound treatment, which, however, finds no support in the data as chlorophyll-*a* concentrations (digitally extracted from figure 4 in Lee et al. 2002), yielded 81 (±56) µg L⁻¹ before and 74 (±42) µg L⁻¹ during ultrasound. The intensity in those studies would be around 5.5 × 10⁻⁹ W mL⁻¹ (in Lee et al. 2002) and 7 × 10⁻⁸ W mL⁻¹ (in Ahn et al. 2007). Such intensities are much lower than the intensities applied in laboratory studies (mean ± 1 SD:

Table 1 Overview of plants of which extracts (solvent given) have been tested on target cyanobacteria, including reported effects

Plant	Target	Extract	Test	Effect	Reference
<i>Solidago canadensis</i> L.	<i>M. aeruginosa</i>	Ethanol (no ethanol control)	Laboratory	At 0.3–0.5 g L ⁻¹ >90 % inhibition	Huang et al. (2013)
<i>Pistia stratiotes</i>	<i>M. aeruginosa</i>	Methanol, ethyl acetate and <i>n</i> -hexane	Laboratory	20–500 mg L ⁻¹ ; growth inhibition EC _{50,7d} = 80–210 mg L ⁻¹ , recovery after 7 d	Wu et al. (2013)
<i>Myriophyllum aquaticum</i>	<i>M. aeruginosa</i>	Water	Laboratory	Growth inhibition	Wu et al. (2008)
<i>Chara australis</i>	<i>A. variabilis</i>	60 % methanol	Laboratory	Clearing effect	Pakdel et al. (2013)
<i>Potamogeton crispus</i>		Live plants, exudates			
Barley straw	<i>Aphanizomenon flos-aquae</i>	water	Laboratory	1–5 g L ⁻¹ ; growth inhibition 2.5 g L ⁻¹ ; growth inhibition	Haggard et al. (2013)
75 % <i>Scirpus acutus</i> —25 % <i>Typha latifolia</i>					
<i>Salvia miltiorrhiza</i>	<i>M. aeruginosa</i>	Ethyl acetate	Laboratory	1.25–20 mg L ⁻¹ ; growth inhibition EC ₅₀ = 4.68 mg L ⁻¹	Zhang et al. (2013)
<i>Acacia mearnsii</i>	<i>M. aeruginosa</i> Natural community	Not specified	Laboratory, field mesocosms	3.623 mg L ⁻¹ ; max 59 % reduction 1–4 mg L ⁻¹ ; strong growth reduction ~28 % of cell density of control bloom	Zhou et al. (2012)
<i>Ephedra equisetina</i>	<i>M. aeruginosa</i> Natural community	Water	Laboratory, ponds	50–375 µg L ⁻¹ ; decline CHL 1.25 mg L ⁻¹ ; rapid decline in CHL, recovery after 1 week	Yan et al. (2012)
<i>Myriophyllum verticillatum</i>	<i>M. aeruginosa</i>	Tannic acid water	Laboratory	1–5 g L ⁻¹ ; growth inhibition, but stimulation in presence of <i>Desmodesmus</i>	Chang et al. (2012)
<i>Phragmites australis</i> , <i>Carex dispalata</i> , <i>Typha domingensis</i>	<i>M. aeruginosa</i>	Water	Laboratory	unspecified dose; growth inhibition	Takeda et al. (2011)
<i>Eichhornia crassipes</i>	<i>Spirulina platensis</i> , <i>Nostoc piscinale</i>	Methanol	Laboratory	20–250 mg L ⁻¹ ; no effect	Shanab et al. (2010)
<i>Pururus yedoensis</i> , <i>Acer buergerianum</i>	<i>M. aeruginosa</i>	Water, casein/acetone, ethanol 50 %	Laboratory	2.2–26 pg cell ⁻¹ ; decline in cell density	Shimada et al. (2010)
<i>Acacia mimosa</i>	<i>M. aeruginosa</i>	Not specified	Laboratory	2–12 mg L ⁻¹ ; growth inhibition, max ~50 %, recovery occurs	Zhou et al. (2010)
<i>Moringa oleifera</i>	<i>M. aeruginosa</i>	Water	Laboratory	1.6–160 mg L ⁻¹ ; growth inhibition >20 mg L ⁻¹ , recovery observed	Lüring and Beekman (2010)

Table 1 continued

Plant	Target	Extract	Test	Effect	Reference
<i>Stratiotes aloides</i>	<i>Anabaena variabilis</i> , <i>Merismopedia tenuissima</i> , <i>Leptolyngbya boryana</i>	50 % methanol	Laboratory	0.1–4 g L ⁻¹ Growth inhibition <i>A. variabilis</i>	Mohamed and Shehri (2010)
66 Chinese medicinal herbs	<i>M. aeruginosa</i>	Water boiled	Laboratory	No effect on <i>M. tenuissima</i> and <i>L. boryana</i> 16 extracts inhibited growth Minimum inhibitory concentration 0.39–25 g L ⁻¹	Yang et al. (2009a)
<i>Oryza sativa</i>	<i>M. aeruginosa</i>	Methanol, ethyl acetate	Laboratory	0.01–10,000 µg L ⁻¹ ; max 64 % growth inhibition	Park et al. (2009)
<i>Papaveraceae</i>	<i>M. aeruginosa</i>	Water boiling	Laboratory	EC ₅₀ : 56–871 mg L ⁻¹	Jančula et al. (2007)
<i>Stratiotes aloides</i>	<i>Synechococcus leopoliensis</i> <i>M. aeruginosa</i> <i>Anabaena</i> sp.	Water, methanol, acetone	Laboratory	EC ₅₀ : 46–985 mg L ⁻¹ 0.5–3.75 g L ⁻¹ ; inhibition >1 g L ⁻¹ , stimulation at 0.75 g L ⁻¹	Mulderij et al. (2007)
<i>Myriophyllum verticillatum</i>	<i>Anabaena</i> sp.	50 % methanol	Laboratory	0.5–2 mg; clearing area found	Hilt et al. (2006)
<i>Elodea nuttallii</i>	<i>E. canadensis</i>	Methanol		No effect on CHL	Erhard and Gross (2006)
Rice straw	<i>An. variabilis</i> <i>Synechococcus elongatus</i> Syn. sp. <i>Syn. nidulans</i> <i>Pseudanabaena cf. catenata</i> <i>M. aeruginosa</i>	Methanol	Laboratory	0.5–2 mg; clearing area found 0.5–2 mg; clearing area found 0–1 g L ⁻¹ ; growth reduction 56–92 %	Park et al. (2006)
<i>Stratiotes aloides</i>	<i>M. aeruginosa</i>	Spent medium	Laboratory	0.01–10 mg L ⁻¹ ; growth reduction 98 % Increased lag phase, slightly higher growth	Mulderij et al. (2005)

$0.115 \pm 0.084 \text{ W mL}^{-1}$, $n = 26$, 20/40 kHz; Al-Hamdani et al. 1998; Francko et al. 1990; 1994; Hao et al. 2004a, b; Joyce et al. 2010; Rajasekhar et al. 2012b; Thomas et al. 1989; Zhang et al. 2006; Wu et al. 2012; Rodriguez-Molares et al. 2014).

Intensities of most, if not all, devices that are being sold for clearing lakes, ponds and aquaria are low. Some manufacturers even pointed out that for their commercial available transducers “the occurrence of cavitation can be disregarded” (URL7), or “the method is not cavitation” (URL8). The supposed mode of action “is purely based on killing algae by bringing them in resonance” (URL9). For cyanobacteria, the underlying assumption is that ultrasound will cause resonance and subsequent rupture or collapse of gas vesicles (Rajasekhar et al. 2012a). The resonance frequency (f_0) of gas bubbles can be estimated with the equation (Kotopoulis et al. 2009):

$$f_0 = \frac{1}{2\pi} \sqrt{\left(\frac{3\gamma}{R_0^2 \rho} \left(p_0 + \frac{2\sigma}{R_0} + \frac{2\chi}{R_0} \right) - \left(\frac{2\sigma + 6\chi}{R_0^3 \rho} \right) \right)} \quad (1)$$

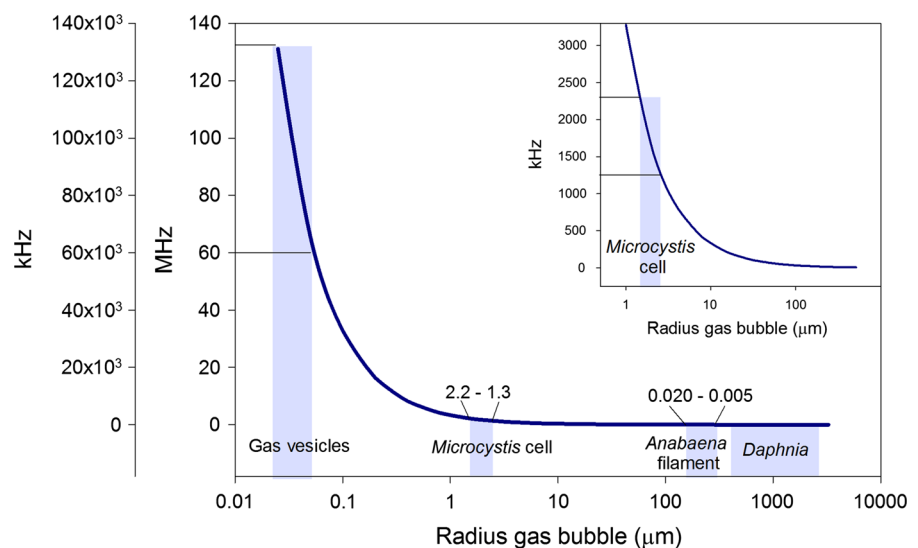
in which γ is the polytropic exponent of the gas (1.39 for air), R_0 is the radius of the bubble (μm), ρ is the density of the surrounding liquid (1000 kg m^{-3}), p_0 is the ambient pressure (10^5 Pa), σ is the surface tension of the surrounding medium (Nm^{-1}), and χ is the membrane elasticity (Nm^{-1}). The surface tension and membrane elasticity were taken as in Kotopoulis et al.

2009; 0.072 and 0.044 Nm^{-1} , respectively. With this equation (Eq. 1), the resonance frequency can be calculated (Fig. 3). As a rule of thumb, the resonance frequency (in MHz) can also be approximated by $6.5/\text{bubble diameter in } \mu\text{m}$ (pers. comm. M. Postema, prof. in acoustics, University of Bergen, Norway).

Equation 1 follows that a sphere with a diameter of $5 \mu\text{m}$ would require 1.3 MHz, a $1\text{-}\mu\text{m}$ sphere 6.5 MHz, a $0.6\text{-}\mu\text{m}$ sphere 11 MHz, a $0.1\text{-}\mu\text{m}$ sphere 65 MHz and a 60-nm sphere 109 MHz to bring them into resonance. Here, the shape of the gas vesicles is assumed to be spherical, but in reality they have the form of a hollow cylindrical tube (Walsby and Hayes 1989). Gas vesicles in *M. aeruginosa* have a diameter of $60\text{--}70 \text{ nm}$ and maximum length of around 600 nm (Walsby 1994; Dunton and Walsby 2005), which means that it is highly unlikely that low-frequency ultrasound ($\sim 20\text{--}200 \text{ kHz}$) from such commercial low-power systems will provoke resonance of gas vesicles and subsequent collapse of gas vesicles in the cyanobacteria. However, acoustic cavitation may cause gas vesicle damage (Lee et al. 2001; Rodriguez-Molares et al. 2014), while in the acoustic field near the transducers a high acoustic pressure (power) will kill everything, not only cyanobacteria, but as mentioned before effective control of cyanobacteria in lakes and ponds by such devices is highly questionable.

Also some studies that had used higher-frequency ultrasound (1700 kHz) contain some peculiarities,

Fig. 3 Calculated resonance frequencies (using Eq. 1) of gas bubbles varying in radius (μm). Also included are the ranges for cyanobacteria gas vesicles, *Microcystis* cell, an *Anabaena* filament and *Daphnia*



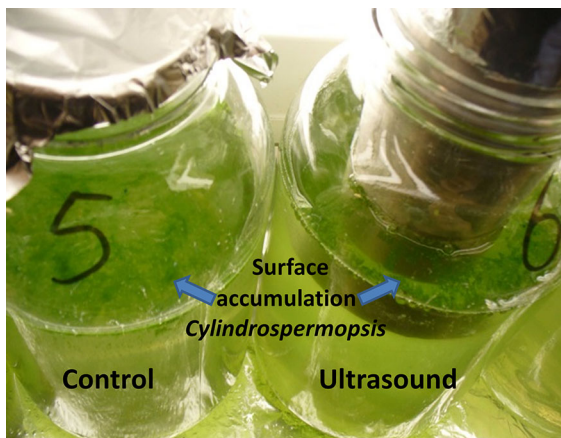


Fig. 4 Jars containing 800 mL *Cyindrospermopsis raciborskii* suspension after 10 days of exposure to ultrasound (6) where the surface accumulation is comparable to the control (5)

which make the suggested gas vesicles resonance doubtful; Tang et al. (2004) derived a resonance frequency of “1.30–2.16 MHz because the R of the cyanobacteria vacuoles was in the range of 3–5 μm ,” while Hao et al. (2004b) mentioned that “the gas vesicles are usually up to 1 μm in length.” This seems a rather large exaggeration of gas vesicle sizes.

Absence of ultrasound-induced gas vesicle rupture is supported by a controlled experiment with Flexidal AL-10 transducers (Lürling and Tolman 2014a), in which there was not only no wipe out of *Cyindrospermopsis raciborskii*, but also no effect on buoyancy (Fig. 4). Similarly, *Anabaena* sp., *Microcystis aeruginosa* and the green alga *Scenedesmus obliquus* could not be controlled by these transducers that produced ultrasound at 20, 28 and 44 kHz with an acoustic power of 0.7 W (Lürling and Tolman 2014a; Lürling et al. 2014). The experiments unequivocally demonstrated

that these devices were not able to clear even small volumes (800 mL—intensity of $8.5 \times 10^{-4} \text{ W mL}^{-1}$) of cyanobacteria or chlorophytes thereby refuting the manufacturers claim that “phytoplankton would be killed within one week” (URL10). However, the zooplankton grazer *Daphnia magna* was killed within 15-min exposure (Lürling and Tolman 2014a). In a follow-up using Flexidal AL-05 transducers, ultrasound was not able to clear 85-L tanks over a 25-day experimental period (Lürling and Tolman 2014b). Six tanks were inoculated with a mixture of green algae and cyanobacteria and stocked with some zooplankton grazers (*Daphnia*). While in controls the *Daphnia* strongly suppressed the phytoplankton creating clear water after 3 weeks, the ultrasound treatments turned into a green phytoplankton-dominated soup (Fig. 5). This was caused by a strong detrimental effect of ultrasound on *Daphnia* as was demonstrated in additional experiments (Lürling and Tolman 2014b).

The findings that non-target organisms, such as *Daphnia*, can be killed by ultrasound from the commercial available transducers are in direct conflict with the claim that ultrasound is “environmental friendly” (Rajasekhar et al. 2012a). Actually, such claim and that ultrasound can be considered a “green solution” (Wu et al. 2011) find no support in the literature. None of the studies reviewed in Rajasekhar et al. (2012a) and Wu et al. (2011) included controlled experiments to examine the effect of ultrasound on non-target organisms such as *Daphnia*. High-power ultrasound is also used for disinfection of ballast water or raw water for drinking water preparation, where it may inactivate motile plankton (Hoyer and Clasen 2002) or kill zooplankton, especially larger

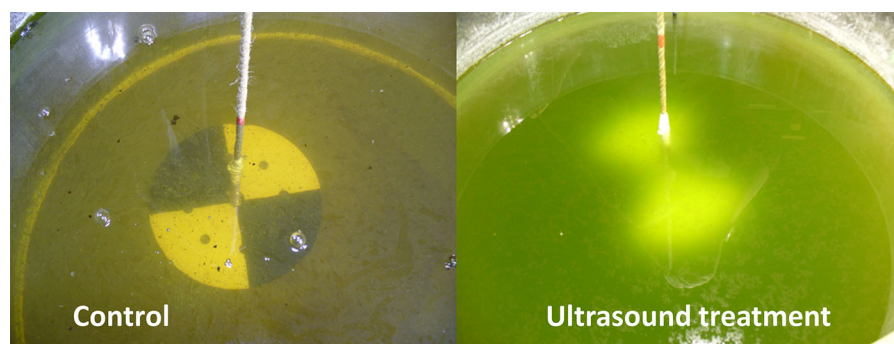


Fig. 5 Pictures after 3-week incubation of a non-exposed (control) and ultrasound-treated 85-L tank (ultrasound treatment)

cladocerans (Holm et al. 2008). Cavitation might damage fish skin (Frenkel et al. 1999), while detrimental effects of ultrasound on macrophytes have been reported (Carstensen et al. 1990; Wu and Wu 2006). Hence, high ultrasound intensities might come with danger for non-target organisms in the vicinity of the transducers.

The commercially available ultrasound devices had no water-clearing effect in relatively small volumes of 800 mL and 85 L (Lüring and Tolman 2014a,b; Lüring et al. 2014). Likewise, field trials with comparable devices that have been conducted in the Netherlands in 2007, yielded no evidence of an effect of ultrasound on cyanobacteria or phytoplankton (Govaert et al. 2007; Kardinaal et al. 2008). The study of Govaert et al. (2007) was conducted in two identical ponds of which one was treated with ultrasound produced by a Flexidal AL-50 transducer, while the other one served as control. During the four months of operation, chlorophyll-*a* concentrations in the control were around $64 (\pm 13) \mu\text{g L}^{-1}$ and in the ultrasound treatment around $69 (\pm 26) \mu\text{g L}^{-1}$ (data digitally extracted from Fig. 2 in Govaert et al. 2007). Moreover, no difference in phytoplankton composition was found (Govaert et al. 2007). Kardinaal et al. (2008) described two other field trials in the Netherlands, one in the southwest of the Netherlands in a harbor area near Tholen and the other one in a bay of recreational area De Gouden Ham near the river Maas. Surface scums and high *Microcystis* densities were observed on both sites despite the ultrasound treatment, and the authors concluded that ultrasound was not effective in reducing cyanobacteria (Kardinaal et al. 2008).

In analogy to what has been reported for bacteria (Joyce et al. 2003; Mason 2007), the effects of ultrasound on cyanobacteria can be grouped as:

- High-intensity (power) ultrasound in small volumes kills cyanobacteria. The devices are predominantly those that are meant for cleaning and sterilization.
- High-intensity (power) ultrasound in large volumes has no lethal effect on cyanobacteria. Filament breakage and colony declumping may occur.
- Low-intensity (power) ultrasound in small volumes does not kill cyanobacteria, but causes filament breakage and some growth reduction.

- Low-intensity ultrasound in large volumes has no effect on cyanobacteria.

The use of artificial mixing devices in non-stratifying waters

Whereas the use of artificial mixers in stratifying water has been dealt extensively elsewhere in this issue (Visser et al. 2016), we would like to devote a few words to the use of artificial mixers in non-stratifying waters. In a review on the application of aeration in American reservoirs, Pastorok et al. (1980) show that in only 50 % of the cases, artificial mixing was successful. An average depth >40 m seems to be necessary to create the light conditions that bring the total phytoplankton biomass down (Klapper 1991). In non-stratifying, mixed systems, the added advantage of being buoyant is limited to non-existent. In these shallow systems, mixing can be even counterproductive, by disrupting the sediment surface and enhancing phosphorus release (Blottiere et al. 2014). Indeed, Barbiero et al. showed that artificial mixing in a small, shallow impoundment close to New York (USA) not only did not result in the desired reduction in cyanobacterial dominance, but also led to an increased flux of phosphorus from the sediment, potentially even fueling more biomass buildup (Barbiero et al. 1996a, b). Also in shallow lake Sheldon ($z_{\text{max}} = 3.0$ m), artificial mixing could not mitigate cyanobacterial blooms (Oberholster et al. 2006). Although artificial mixers such as the Solarbee® have been claimed to mitigate water quality problems in shallower waters by taking away the competitive advantage that buoyant cyanobacteria have over other phytoplankton species that cannot control their buoyancy, the failure to reduce nuisance caused by phytoplankton in shallow systems tells another story (Barkoh et al. 2011; Bocchichio 2012; Hudnell et al. 2010). In July 2013, a Solarbee® was installed in the shallow—average 2 m depth—Krabbeplas (the Netherlands) to control cyanobacteria nuisance. However, the already issued swimming ban lasted until beginning September 2013 as cyanobacteria remained flourishing in the lake. For instance, a water sample from August 28, 2013, measured with a PHYTO-PAM revealed $133 \mu\text{g L}^{-1}$ cyanobacterial chlorophyll-*a*. A warning for cyanobacteria in May 2014 was followed by

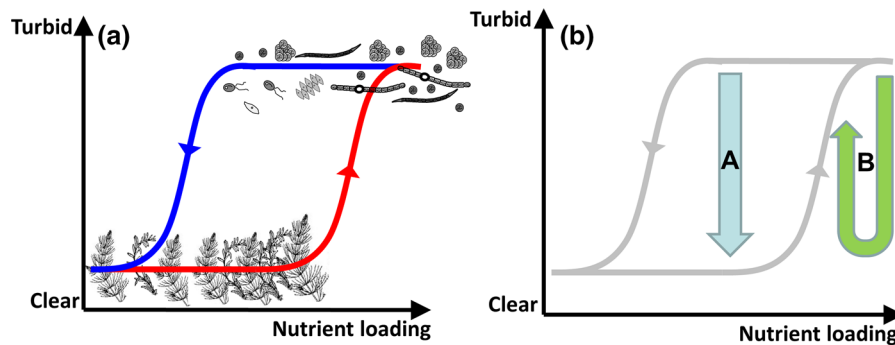


Fig. 6 **a** Hysteresis in a typical shallow lake where increased nutrient loading leads to a phytoplankton-dominated state that can be brought back to a clear water state with submerged macrophytes by strong reduction in nutrient loading. The critical point of return is at lower loading than the transition to a turbid state (*cf.* Scheffer et al. 1993). **b** Shock measures strongly

reducing turbidity like end-of-pipe cyanobactericides will be able to bring a system in a clear water state when applied to a water that is below the critical nutrient loading for turbidity (*arrow A*). However, when applied above the critical loading, the effects will be of short duration and the system will return to a turbid state (*arrow B*)

several warnings and negative bathing advices from June 30, 2014, until September 2014. Hence, the Solarbee[®] was ineffective in controlling cyanobacteria nuisance in this lake.

In conclusion, we advise against artificial mixing of shallower waters, as sediment release of phosphorus may fuel cyanobacterial blooms rather than mitigate nuisance by cyanobacteria.

Conclusions and recommendations

Tackling nutrient inputs and internal loading is generally considered a prerequisite for water quality improvement and long-term reduction in cyanobacteria blooms (Cooke et al. 2005; Hilt et al. 2006), but the required preventive measures are not always feasible (Jančula and Maršálek 2011) or may be insufficient to bring the water body in the desired state, which is often a clear water. In particular, in shallow lakes, a typical hysteresis can be observed (Scheffer et al. 1993) where increased nutrient loading first passes by rather unnoticed, stimulating submerged macrophytes, but once crossed a critical loading, the water body shifts into a phytoplankton-dominated state (Fig. 6). This is often a cyanobacteria-dominated state (Scheffer et al. 1997). Reducing the nutrient loading will express little effect on water clarity, unless reduction is very strong traversing a critical loading at much lower values than the one that caused a turbid state, where after the system returns to a clear water state (Fig. 6a). Between

those transition boundaries, the water body can have two alternative stable states, a turbid phytoplankton-dominated state and a clear water state with submerged macrophytes (Scheffer et al. 1993). Here end-of pipe solutions might be of use when nutrient reduction alone has little impact on water clarity (Fig. 6b, arrow A) and a disturbance with a strong water-clearing agent can bring the water back to a stable clear state (Scheffer et al. 1993).

Hence, increased water transparency is one of the desired effects after fast reduction in the cyanobacteria biomass, but the curative products should also improve the water quality long enough to allow submerged macrophytes to establish. To determine whether such a shock therapy might be feasible, a decent system analysis on nutrient loadings is crucial, because interceptions above the critical loading for the transition from clear to turbid (Fig. 6b, arrow B) will only be short-lived and the system will return to its turbid, cyanobacteria-dominated state.

To our opinion, the road to more evidence-based mitigation of cyanobacterial blooms should always start with a system analysis of the specific water system. This implies a thorough investigation of its water and nutrient flows, the biological makeup of the system and the societal environment related to the functions of the specific water. There is broad consensus that nutrient enrichment leads to harmful cyanobacterial blooms (e.g., Paerl et al. 2011) and thus determining the nutrient inputs is a first logical step (Cooke et al. 2005). The nutrient flows separate in

external load, water and non-water-related and internal load. In deep lakes, acceptable loadings can be derived from empirical relationships (e.g., Vollenweider 1976). The overall nutrient loading for shallow lakes can be evaluated by using the model PCLake (Janse et al. 2008) that will indicate the critical nutrient loads for clear to turbid water and vice versa for the specific water bodies. In relation to the biological makeup, it is crucial to determine whether a water body is packed or not with lake-bottom resuspending fish (carps, breams). If those fish are abundant in densities of hundreds of kg per hectare, the water will remain in a turbid state by resuspending sediments and preventing submerged macrophyte establishment (e.g., Cline et al. 1994; Roozen et al. 2007). In such cases, fish removal—maybe in combination with macrophyte introduction—should be considered an essential structural measure for rehabilitation. Besides efficiency and applicability, lake managers also need to consider safety, costs and societal support of proposed measures. Consequently, the system analysis will lead to tailor-made solutions and most probably a set of measures rather than one single measure, but also leaving doing nothing as an option (Mackay et al. 2014).

The problem of cyanobacteria blooms and nuisance might be evident and may press water authorities to undertake immediate actions from obvious warnings to even trial and error activities with all kind of “miracle” techniques and products. In that context, numerous products have been proposed to Dutch water authorities as end-all solutions in controlling cyanobacteria. In this review, we have critically evaluated the claims and effectiveness of “effective” microorganisms, golden algae, plant extracts, ultrasound and artificial mixers in non-stratifying lakes. These were selected because of strong promotion or media attention the products/techniques received in the Netherlands over the last decade and of which information is available. There are many more or less comparable products/techniques that are tried by water authorities (e.g., “Dango-balls” URL11, “vortex-system” URL12, “oxatur” URL13, “oil screens” URL14) that have been left out of the evaluation, but of which efficacy is likewise controversial. In general, proposed underlying mechanisms are doubtful. The “effective” microorganisms do not outcompete cyanobacteria, and there is no reason to expect they will improve water quality through diminished

cyanobacteria/phytoplankton abundance. Golden algae cannot consume large cyanobacteria, and despite their omnipresence, they apparently fail to control blooms, but even if they did the resulting golden algae bloom would still keep water turbid. Plant extracts are needed in high quantities, might have short-lived effects, might have rather long lag phase and could come with negative side effects such as exacerbated eutrophication, lower oxygen levels or release of cyanotoxins, bringing too much uncertainties to warrant application. Commercially available ultrasound transducers emit frequencies that cannot collapse gas vesicles, and in contrast to the manufacturer’s claim, they do not clear the water, but are harmful to other aquatic organisms, at least to *Daphnia*. The “positive” laboratory studies have all used high-power devices that are designed for cleaning and sterilization through disruption of cells, bacteria, spores or tissue. These devices cannot be used in situ, and even if they would, the much larger volumes would mean a strongly reduced power transmission and thus diminished impact on cyanobacteria. Hence, there is no music in fighting cyanobacteria with ultrasound. Finally, mixing can have positive effects in deeper, stratifying waters (see Visser et al. 2016), but mixing of shallower waters should be avoided as sediment release of phosphorus may fuel cyanobacterial blooms rather than mitigate nuisance by cyanobacteria. Therefore, none of the above seem the wide applicable solution to cyanobacterial nuisance; they should not be considered Columbus’s egg.

Nonetheless, in addition to eutrophic lakes and ponds also in oligotrophic and mesotrophic lakes and ponds, cyanobacteria might still be an important component of the plankton community (Lepistö et al. 2005; Carey et al. 2012). Even very low concentrations of cyanobacteria can become positively buoyant, accumulate at the water surface and be further concentrated on leeside shores. In such systems, reduction in nutrient loading is not the measure of first choice, while mechanical removal, sedimentation or killing of the accumulated material seem more feasible. The latter could be achieved with selected plant extracts (Table 1) that comes with abovementioned drawbacks or hydrogen peroxide (Matthijs et al. 2012; Jančula et al. 2016). Whether these will also be applicable when cyanobacteria scums have accumulated in harbors or in the vicinity

of houses giving strong nuisance mainly through foul odors remains to be seen. Then probably skimming of cyanobacteria off the surface or sinking the intact cells with a coagulant and ballast (e.g., Li and Pan 2013; Lürling and Van Oosterhout 2013) might offer an alternative. There is a wealth of information on the latter technique, where natural soils and clays are modified with flocculants to effectively remove cyanobacteria from the water column (Noyma et al. 2015; Pan et al. 2006a, b, 2011a, b; Zou et al. 2006). An in situ experiment in Lake Taihu applying 25–31 mg L⁻¹ (40–50 g m⁻²) effectively cleared an isolated bay of cyanobacteria (Pan et al. 2011a). Inasmuch as in this technique entrapped cyanobacteria in flocks remain intact (Chow et al. 1999; Drikas et al. 2001), no release of toxins and nutrients during treatment occurs. Hence, the stripping of the water column from cyanobacteria might provide a promising alternative to the use of algacides, such as copper sulfate or hydrogen peroxide, that may come with shortcomings such as toxins and nutrients release (Merel et al. 2013). In stratifying waters, cyanobacteria removal with flocculants and modified clay has yielded promising results too (Lürling and Van Oosterhout 2013; Waajen et al. 2015). Nonetheless, more research on prevention of resuspension or liberation of viable cells from flocks in shallow waters is needed, where inclusion of calcium peroxide pellets (Noyma et al. 2015) or toxin-degrading bacteria (Li and Pan 2015) may further improve performance.

Overall, water managers display a great need for effective treatments to control eutrophication and mitigate cyanobacterial nuisance. In order to solve the problems and to select the most promising set of measures, identification of the cause(s) is crucial. Based on the outcome of the essential system analysis, water managers may choose efficient, easy, safe and cheap measures from the tool box that ideally contains numerous preventive and curative measures. However, this critical review clearly showed that there is no strong support for several end-of pipe measures, such as “effective” microorganisms, golden algae, plant extracts, ultrasound and artificial mixers in non-stratifying systems to diminish eutrophication problems such that the resulting water quality meets societal and legislation demands. In that view, rapid interventions through flocking and sinking cyanobacteria with a coagulant and ballast seem more

promising and further research should be devoted to these encouraging techniques.

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Compliance with ethical standards

Conflict of interest The authors declare no conflict of interest.

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