



HAL
open science

Development of environmental loop-mediated isothermal amplification (eLAMP) diagnostic tool for *Bulinus truncatus* field detection

M. Blin, Bruno Senghor, Jérôme Boissier, Stephen Mulero, Olivier REY,
Julien Portela

► To cite this version:

M. Blin, Bruno Senghor, Jérôme Boissier, Stephen Mulero, Olivier REY, et al.. Development of environmental loop-mediated isothermal amplification (eLAMP) diagnostic tool for *Bulinus truncatus* field detection. *Parasites & Vectors*, 2023, 16 (1), pp.78. 10.1186/s13071-023-05705-4 . hal-04012029

HAL Id: hal-04012029

<https://hal.science/hal-04012029>

Submitted on 2 Mar 2023

HAL is a multi-disciplinary open access archive for the deposit and dissemination of scientific research documents, whether they are published or not. The documents may come from teaching and research institutions in France or abroad, or from public or private research centers.

L'archive ouverte pluridisciplinaire **HAL**, est destinée au dépôt et à la diffusion de documents scientifiques de niveau recherche, publiés ou non, émanant des établissements d'enseignement et de recherche français ou étrangers, des laboratoires publics ou privés.



Distributed under a Creative Commons Attribution 4.0 International License

Development of environmental loop-mediated isothermal amplification (eLAMP) diagnostic tool for *Bulinus truncatus* field detection

Manon BLIN ^{1,2*}, Bruno SENGHOR ³, Jérôme BOISSIER ¹, Stephen MULERO ^{1,4}, Olivier REY ¹ and Julien PORTELA ^{2*}

1 Hosts Pathogens Environment Interactions, UMR 5244, CNRS, IFREMER,UM, University of Perpignan, Via Domitia, 66860 Perpignan, France

2 SAS ParaDev®, 66860 Perpignan, France

3 VITROME, IRD-UCAD International Campus, 1386 Dakar, Senegal

4 Univ. Grenoble-Alpes, Univ. Savoie Mont Blanc, CNRS-LECA,38000 Grenoble, France

ABSTRACT

Background Global changes are reshaping the distribution of vector-borne diseases by spreading vectors to previously non-endemic areas. Since 2013, urogenital schistosomiasis has emerged in Corsica and threatens European countries. Gastropod vectors release schistosome larvae that can infect humans who come into contact with freshwater bodies. Monitoring schistosomiasis host vectors is a prerequisite to understand and subsequently to control this pathogen transmission. Because malacological surveys are time consuming and require special expertise, the use of a simple molecular method is desirable.

Methods The aim of this study is to develop a ready-to-use protocol using the LAMP (loop-mediated isothermal amplification) method to detect environmental DNA of *Bulinus truncatus*, vector of *Schistosoma haematobium*. Interestingly, LAMP method possesses all the characteristics required for adaptability to field conditions particularly in low-income countries: speed, simplicity, lyophilized reagents, low cost and robustness against DNA amplification inhibitors. We have tested this new method on Corsican water samples previously analysed by qPCR and ddPCR.

Results We demonstrate that our diagnostic tool *B. truncatus* eLAMP (Bt-eLAMP) can detect the eDNA of *Bulinus truncatus* as effectively as the two other methods. Bt-eLAMP can even detect 1/4 of positive samples not detectable by qPCR. Moreover, the complete Bt-eLAMP protocol (sampling, sample pre-process, amplification and revelation) does not require sophisticated equipment and can be done in 1 ½ h.

Conclusions LAMP detection of environmental DNA provides large-scale sensitive surveillance of urogenital schistosomiasis possible by identifying potentially threatened areas. More generally, eLAMP method has great potential in vector-borne diseases and ecology.

KEYWORDS

Environmental DNA, LAMP assay, Intermediate host control, *Bulinus truncatus*

Background

It is well established that global changes increase the risk of infectious disease outbreaks [1–3]. The successive epidemic waves over the past decade such as the swine flu pandemic, numerous Ebola virus outbreaks in West Africa, 2015 Zika virus outbreak and the COVID-19 pandemic all resulted in significant mortality and morbidity while spreading to multiple countries [2]. The incidences of vector-borne diseases in humans and animals are impacted by climate change and human lifestyle (e.g. migration, pollution, urbanization) [1, 4, 5]. For several years, many of them have been emerging or re-emerging worldwide. For example, the geographical range of tickborne encephalitis virus and the human incidence are increasing throughout Europe, including France, in areas that were previously non-endemic [6]. Although schistosomiasis is a disease linked to poverty and lack of potable water in households, schistosome infection has been identified in southern Europe (France and Spain) [7, 8]. Gastropod vectors release schistosome larvae that can infect humans who come into contact with freshwater bodies. In summer 2013, > 106 cases were diagnosed in Corsica, a French Mediterranean island known for its touristic attractiveness [9, 10]. Since then, a few cases of autochthonous transmission have persisted year after year [11]. For many parasitic diseases, global changes lead to a spatial redistribution of the vectors or the intermediate hosts which transmit the diseases and may also lead to disease emergence [12–16]. Mapping infectious disease outbreak areas is essential to both identify populations at risk of infection and communicate and warn the public [17]. Concerning vector-borne diseases, since the transmission is dependent on the presence of the vectors, mapping their spatial distributions is necessary for disease control. It has been shown in several African countries that snail control campaigns play a major role in controlling snail-borne diseases (SBDs) and that regular application of molluscicides probably contributes to the elimination of the SBDs in at-risk areas [18–20]. However, the monitoring of SBDs, particularly those transmitted via freshwater snail species, is less obvious. The spatial and temporal heterogeneity of the distribution of freshwater molluscs explains the difficulty of establishing these risk maps [21, 22]. Conventional sampling methods are therefore laborious and time-consuming, and they require malacologists for taxonomic identification, which have become increasingly scarce in recent years as molecular biology has taken off. Therefore, it is currently very difficult to monitor snail populations on a large spatial and temporal scale [21, 22].

The development of tools to simplify the monitoring of SBDs has been considered to address this issue, based on environmental DNA (eDNA) [23]. eDNA amplification methods were developed to detect many parasite vectors such as *Aedes albopictus* [24] and also snail hosts such as *Galba truncatula* and *Austropeplea tomentosa* [25, 26], both species being involved in the transmission of the liver fluke (*Fasciola hepatica*). The detection of eDNA is usually done using quantitative PCR DNA amplification technique. More recently, the eDNA detection of *Oncomelania hupensis* and *Bulinus truncatus*, involved in the transmission of *Schistosoma japonicum* and *Schistosoma haematobium*, respectively, has been made possible by either quantitative PCR (qPCR) or digital droplet PCR (ddPCR) [21, 27]. The results obtained in these last studies showed high-sensitivity and -specificity detection. Detection of *B. truncatus* eDNA by ddPCR allowed finding up to 0.06 copies/ μ l [21]. This represents a significant advance in the development of tools to simplify SBD monitoring. However, all these techniques require sophisticated and expensive equipment for both sample preparation (i.e. the use of expensive and time-consuming DNA extraction kits to obtain eDNA without PCR inhibitors) and amplification equipment (i.e. LightCycler or ddPCR). These costly techniques require a well equipped laboratory, which is rarely available in schistosome-endemic areas. Development of cost-effective and field-adapted tools for realization of snail distribution maps is still needed [17, 23, 28]. The loop-mediated isothermal amplification (LAMP) technique has the potential to make this possible [29–31]. Interestingly, the LAMP method possesses all the characteristics required for adaptability to field conditions particularly in low-income countries: speed, simplicity, lyophilized reagents, low cost and robustness against DNA amplification inhibitors [29, 30, 32]. Indeed, using a *Bst* polymerase enzyme with strand displacement activity and four to six primers hybridizing on six to eight regions of the target sequence, the exponential DNA amplification reaction can be performed at constant temperature, without prior denaturation of the DNA double strands. Indeed, the hybridization of the primers will

create stem-loop structures on the newly amplified strands leading to the exponential multiplication of the number of new amplification start zones. We finally obtain a smear composed of several fragments of varied sizes which form a ladder after revelation by electrophoresis gel. Thus, the reaction is performed very quickly (usually < 1 h) at a constant temperature of around 63 °C in a simple heating block [29, 31, 33]. Since no large or expensive equipment is required to perform a LAMP, recent studies have focused on the development of portable devices to perform field diagnosis as close as possible to the sampling site [34–37]. Due to its robustness against inhibitors, it can be performed on samples with very simplified preparation, without the need for DNA extraction kits to purify the DNA, which can also be done in the field [38, 39]. DNA amplification can be detected by naked-eye visualization, gel electrophoresis or real-time fluorescence, offering flexibility of use in field settings [34, 36]. Recent studies have shown that it is possible to detect eDNA by LAMP [40–43]. An environmental LAMP (eLAMP) was developed to detect the presence of faecal indicator bacteria in water in 1 h, without sophisticated laboratory equipment or highly trained personnel [40]. Two teams succeeded in amplifying mollusc eDNA (*Galba truncatula* and *Dreissena* sp.) from water samples with LAMP [41, 42].

The aim of this study is to develop a rapid diagnostic LAMP tool to detect the environmental DNA of *Bulinus truncatus*, intermediate host of *Schistosoma haematobium*. Schistosomiasis is the second most common parasitic disease after malaria and remains a neglected tropical disease. Despite its high incidence (230 million infected people and 200,000 deaths per year [44]), efforts to conduct snail control campaigns are very low [28, 45, 46]. However, in 2017, WHO called for refocusing on snail control to sustain progress, calling on member states to develop or adapt national vector control strategies, aiming to reduce the incidence of vector-borne diseases, including schistosomiasis, by at least 40% by 2025 [46]. Thus, cost-effective, field-appropriate tools for rapid mapping of snail distribution are needed for this disease [23]. Compared to qPCR or ddPCR amplification methods, the eLAMP seems advantageous for field applicability in low-income countries. However, this advantage disappears if the DNA extraction protocol that precedes amplification is not adapted to field conditions like using DNA extraction kits. Our work consisted in (i) development of *B. truncatus*-specific eLAMP assay, (ii) comparison of the results obtained by LAMP on an eDNA bank with those obtained by qPCR and ddPCR and (iii) development of a full field-friendly protocol of detection.

Methods

Material

The *Bulinus truncatus* eLAMP (Bt-eLAMP) diagnostic tool was developed from both an environmental DNA (eDNA) bank and newly extracted DNA from molluscs.

The eDNA bank is composed of 200 samples previously collected on Corsica island, France [21]. These samples were DNA extracted from water filtration membranes collected in two distinct rivers from southern Corsica. Urogenital schistosomiasis transmission has recently been evidenced in these two rivers [9, 10, 47]. Three different volumes of water (1, 3 and 5 l) from both the shore and the streambeds of the rivers were collected. Four sites were sampled in the Cavu River: the Mulinu bridge and Tyroliana park sites (sampled in duplicate), the three pools site where *B. truncatus* are present and the water intake site where *B. truncatus* are absent. A single site was sampled from the Solenzara River where *B. truncatus* are present. Negative controls were made by filtering pure spring water along the river. In addition to these sampling sites in Corsica, eDNA was also sampled from an artificial channel within the campus of Perpignan University, where *B. truncatus* has never been recorded, as a negative control. The eDNA extraction protocol from the resulting water filtration membranes was previously described [21] and is briefly recalled here: each filtration membrane was divided into four quarters and extracted using DNeasy® Blood & Tissue Kit (QIAGEN). From the 200 samples, 144 eDNA samples were collected where *B. truncatus* were present and 56 eDNA samples collected where *B. truncatus* were not present, and they were amplified using qPCR. A sample was considered positive if at least one quarter membrane was positive. The ddPCR reactions were run on pools of DNA extracts from the four quarters of each membrane (i.e. 50 reactions) [21]. Results per membrane obtained with both qPCR and ddPCR were compared to eLAMP (present study). Results per membrane quarter obtained by qPCR were also compared to eLAMP.

In addition to eDNAs, and to test the specificity of our primer sets, DNA from freshwater snails (*Bulinus* sp. And other phylogenetically related genera) was also extracted using the DNeasy® Blood & Tissue Kit (QIAGEN) following the tissue extraction protocol. Briefly, after grinding each snail, the samples were put in a Speedvac™ DNA 130 (Savant™, Thermo Scientific™) at 65 °C for 20 min to remove any alcohol or water residuals. The snails were then lysed at 56 °C for 4 h in a solution containing 20 ml proteinase K and 180 ml ATL buffer. The remaining steps were performed according to the manufacturer's instructions. The DNA concentration was measured using a Qubit® 2.0 fluorometer. These last DNAs were diluted with ultrapure water to obtain final concentrations of 0.5 ng/μl. The resulting DNA extracts and dilutions were then stored at -20 °C until used.

eLAMP primer set design

The DNA target selection was exhaustive to maximize the chance of success in the development of the eLAMP diagnostic tool. The ITS2; 5,8S; 18S; COI; and 28S *B. truncatus* DNA regions were downloaded from GenBank database (NCBI, [https:// www. ncbi. nlm. nih. gov/ genba nk/](https://www.ncbi.nlm.nih.gov/genbank/); accession numbers: MH361757 for 5,8S; KJ157340 for 18S [48]; KJ157370 for 28S [48]; MT707426 for COI [49] and MG757890 for ITS2 [50]). When available, sequences from other *Bulinus* species were downloaded from GenBank database to develop a *B. truncatus*-specific detection tool. The design of primer sets was carried out using Primer Explorer V5 software (Eiken Chemical Co., Ltd., Japan; [http:// prime rexp lorer. jp/e/](http://primeexplorer.jp/e/)) following the criteria described in "A Guide to LAMP primer designing" ([https:// prime rexp lorer. jp/e/ v5_ manual/index. html](https://primeexplorer.jp/e/v5_manual/index.html)). The sets had to meet the following parameters: length of each primer between 18 and 22 bp; Tm between 64 and 67 °C for F1c/B1c pair; Tm between 59 and 62 °C for F2/B2 and F3/B3 pairs; GC rate between 50 and 65%; dG threshold < -4 kcal/mol for 5' and 3' stability and between 0 and -1 kcal/mol for dimer check. Primer dimer estimation and stability of each primer set were checked online using the "Multiple Primer Analyzer" (Thermo Scientific Web Tool) and OligoAnalyzer™ Tool (IDT™). All the primers selected were synthesized by Eurogentec (Seraing, Belgium).

LAMP reaction

For each designed primer set several amplification assays were performed by adjusting the amplification reaction parameters (i.e. MgSO₄, primers and betaine concentration, reaction time and temperature). The selected protocol was as follows: a total volume of 10 μl containing 1.2 μM of the internal primers FIP and BIP, 0.2 μM of the external primers F3 and B3, 0.4 μM of the LOOP primers LB and LF, 1.0 mM of each dNTP (New England Biolabs), 1X Isothermal Amplification Buffer II reaction buffer [20 mM Tris-HCl, 10 mM (NH₄)₂SO₄, 150 mM KCl, 2 mM MgSO₄, 0.1% Tween® 20, pH 8.8@25 °C (New England Biolabs, UK)], 3 mM additional MgSO₄ (New England Biolabs, UK), 1.2 M betaine and 1 U of *Bst* 2.0 WarmStart DNA polymerase (New England Biolabs, UK) along with 2.5 μl DNA template. Reaction tubes were placed in a battery-operated heating block at 63°C for 45 min followed by an enzyme inactivation phase at 80 °C for 5 min if the results were not visualised immediately after the reaction. Result visualisations were done according to different protocols classically used in LAMP DNA amplification: (i) final point visual detection of fluorescence after adding 2 μl of 1:50 diluted 10,000× concentration of SYBR Green (Invitrogen) (green: positive; orange: negative); (ii) final point visual detection of the LAMP products on 2% agarose gel electrophoresis with 2 μl MIDORI Green Advance (Nippon Genetics) for 50 ml agarose gel, revealed by UVs; (iii) real-time detection by incorporating Sybr-Green 0.25× in the reaction mix and monitoring the amplification in a Genie III portable device (Optigene Ltd).

Specificity and limit of detection

The specificity of all designed primer sets was assessed on DNA templates from 12 freshwater snail species (Table 1), including four specimens of the same genus but different species (i.e. *B. forskalii*, *B. globosus*, *B. umbilicatus* and *B. senegalensis*) and other phylogenetically related genera. *Bulinus truncatus* from five different localities (Table 1) were evaluated to visualise a possible

intraspecific variability. Only the primer set amplifying exclusively *B. truncatus* DNA was kept for the ensuing experiments.

The limit of detection (LOD) was assessed by LAMP assays on DNA from *B. truncatus* tenfold serially diluted from 10 ng/μl to 1.10⁻⁹ ng/μl before and after adding LOOP primers to the reaction. From the detection limit DNA concentration obtained, the number of *B. truncatus* target copies per microlitre was estimated using the following equation:

$$N_c = \frac{C \times V \times 1.10^{-9}}{M \times S_g \times 2} \times N_A \times N_{rDNA}$$

where N_c = the number of *B. truncatus* target copies per microlitre; C = limit detection DNA concentration in ng/μl; V = volume (here, 1 μl); M = average molar mass of a nitrogenous base = 309 g/mol; S_g = *B. truncatus* genome size = 1.2.109 pb; N_A = Avogadro number; N_{rDNA} = average rDNA copy number in a genome = 93 copies [51–56].

Field applicability

We have developed a new field-friendly eDNA concentration protocol allowing us to take full advantage of the field benefits of LAMP. The water is collected in a clean plastic commercial bottle on the surface of the water body. Then, 500 ml to 1500 ml of water is filtered along the river using a 0.45 μm filtration unit (PES VWR 1L 514-0301) and a manual pump. The water is filtered until the membrane is blocked because of sediment stacking. The membrane is cut in four quarters with a scalpel blade and deposited in a single 25-ml Falcon™ tube containing 15 ml lysis buffer [100 mM NaCl, 250 mM EDTA, 5% SDS and 10 mM Tris–HCl (pH = 8)]. Clamps used to remove the membranes from filtration units are decontaminated before each sampling by successively placing them in a 10% bleach bath for 15 s and then 15 s in a DNA AWAY™ solution (Thermo Scientific). After vigorous agitation, the liquid is collected with a 20-ml syringe. The liquid is pre-filtered with a 40-μm membrane to remove the larger sediments and then filtered with a 0.45-μm (13-mm) Whatman® GD/XP syringe filter (PTFE Sigma Aldrich WHA69742504). The last filter is washed with 5 ml distilled water and dried with the same volume of air. The eDNA is finally recovered by reverse filtration with 500 μl ultrapure water thanks to a syringe coupled with a conical 18 GA dispensing needle (Metcal).

This last protocol was assessed in two Senegalese water bodies investigated in July 2022. The Lampsar site (16°06'29"N; 16°20'59"W) and Diama site (16°12'36"N; 16°24'10"W) are both situated along the Senegal River in Saint-Louis region, Senegal. At each site, water samples were collected in triplicate with about 10 m between samplings. Seven hundred millilitres, two times 500 ml and three times 500 ml were collected for Lampsar and Diama sites, respectively. At each site negative controls were carried out by filtering 1500 ml pure spring water strictly following the same procedure as for biological samples. The presence on site of *B. truncatus* was verified by sampling molluscs for 1 h by two malacologists.

Statistical analysis

The agreement between results obtained with the newly developed LAMP and the qPCR for the detection of *B. truncatus* DNA on the 200 field eDNA library were statistically analysed calculating a kappa coefficient analysis using psych package (version 2.2.9) in R software. Field sensitivities on the eDNA bank were calculated with a 95% confidence interval using stats package (version 3.6.2) in R software

Results

Specificity and limit of detection of selected LAMP primer sets

A total of 18 primer sets were designed. After parameter adjustment and sensitivity and specificity assessment, we have selected the best candidate which targets the ITS2" marker (Table 2). An additional file shows the primer alignment on the ITS2 sequence (see Additional file 1). Primer dimer minimum dG for this set was –0.93 kcal/ mol.

Bt-eLAMP specificity was assessed by testing extracted DNA from 11 non-targeted species and five targeted molluscs from different localities. Figure 1a shows that only the five *B. truncatus* DNA samples are amplified. Sensitivity test (Fig. 1b) shows that the LOD is 1 fg/μl (i.e. equivalent to about $N_c = 0.07$ copies per microlitre) with complete primer set (including LOOP primers). The detection limit was 1 pg/μl before adding the LOOP primers.

Comparison of LAMP to qPCR and ddPCR amplification methods on eDNA samples

Figure 2 shows detection accuracy comparison among LAMP, qPCR and ddPCR amplification methods on eDNA samples collected from two Corsican rivers. The Bt-eLAMP method provided the same results as those obtained using the qPCR and ddPCR. All membranes are positive except for the "1LStreambed" membrane from the Mulinu bridge two site. No positive signal was detected from the six field negative controls or from the University site where *B. truncatus* never occurred or for samples collected at Water intake site from the Cavu River where no *B. truncatus* were collected (Fig. 2). Considering results obtained per membrane quarter where *B. truncatus* was present, 110 of 144 samples were successfully amplified with LAMP; meanwhile, 91 of 144 samples were successfully amplified with qPCR. The sensitivities of LAMP and qPCR methods were estimated to be 76.4% (95% CI 68.6–83.1) and 63.2% (95% CI 54.8– 71.1), respectively. Kappa agreement obtained with these results is equal to 0.51 (Table 3).

Field applicability

The use of LAMP amplification as field-friendly detection method was improved by developing a new eDNA concentration protocol that could be done along the water bodies. The entire protocol was tested on Lampsar and Diama sites, Senegal. In these two sites, respectively, 95 and 229 *B. truncatus* were sampled by two malacologists for 1 h sampling. Four of the 95 *B. truncatus* sampled from Lampsar site emitted cercariae after 2 h light exposure. *Bulinus globosus* was present at both sites. *Bulinus forskalii* was only present at Diama site, while we found *Biomphalaria pfeifferi* only at Lampsar site. The three eDNA samplings from the two sites were positive (Fig. 3) showing 100% accuracy of the method. None of the negative controls were positive. Samples of *B. truncatus* eDNA were amplified on average 12 min after amplification of the positive controls (*B. truncatus* extracted DNA).

Discussion

Global changes are reshaping the distribution of vector-borne diseases by spreading vectors to previously non-endemic areas. Therefore, the development of cost-effective and field-adapted tools for vector distribution maps is necessary [17, 28]. In the present study, we have developed a rapid LAMP diagnostic tool to detect the environmental DNA of a freshwater snail vector (i.e. *B. truncatus*) of a tropical parasite (i.e. *S. haematobium*). Our Bt-eLAMP successfully amplified an ITS2 DNA marker fraction of *B. truncatus* after 45 min of incubation at 63 °C. In vitro assays evidenced the high sensitivity of the Bt-eLAMP with a LOD equal to 1 fg/μl when it included LOOP primers. Considering that the LOD was 1 pg/μl before adding the LOOP primers, we deduce that the addition of LOOP primers allows a gain in sensitivity of a factor of 1000. From a methodological point of view, this highlights the importance of designing primer sets allowing inclusion of LOOP primers. The detection limits of the developed LAMP techniques are generally between 100–0.1 fg/μl [43, 57–59]. Our LOD is lower compared to the LOD previously evaluated for the LAMP eDNA detection of the freshwater snail vector of fasciolosis (*Galba truncatula*) [41]. These last authors measured a 0.349 pg/μl LOD [41]. When estimating the copy number equivalence (0.07 copies per microlitre) with the complete primer set (i.e. including LOOP primers), we found a sensitivity close to that obtained using ddPCR (0.06 copies per microlitre) [21]. Our Bt-eLAMP showed equal sensitivity compared to the ddPCR, which is considered the most sensitive DNA amplification technique of all [60, 61].

Our Bt-eLAMP assay showed 100% specificity for *B. truncatus* even considering closely related snail species. This suggests that the Bt-eLAMP assay is robust enough to detect *B. truncatus* eDNA in areas where other snails co-occur without risk of cross-reactivity. Our tool can also allow species

determination between *B. truncatus* and other *Bulinus* species such as *B. globosus*, which are morphologically indistinguishable. On the other hand, the usefulness of this high specificity could be questioned if we consider that some molluscs' sister species, such as *B. globosus*, could also be involved in the transmission of urogenital schistosomiasis [62]. It would be interesting to also develop eDNA diagnostic tools allowing a pan-specific detection of both *B. truncatus* and *B. globosus* or all species of the *Bulinus* genus. The difficulty will be to find an equilibrium between sufficiently conserved regions between *Bulinus* species and sufficiently different from other gastropods to obtain a genus-specific primer set. This last task is more difficult as the number of primers required in LAMP and the positioning constraints between them on the target sequence increase the length of the sequence to be found meeting these requirements.

Considering our eDNA bank, the Bt-eLAMP method provided exactly the same overall detection capacity compared to the qPCR or the ddPCR [21]. Considering each membrane quarter, with 76.4% (95% CI 68.6–83.1) and 63.2% (95% CI 54.8–71.1) of positive detection using LAMP or qPCR, respectively, the LAMP method showed higher detection capacity compared to qPCR [21]. The kappa coefficient highlights moderate agreement between the two methods. Fifteen membrane quarters are negative in LAMP while they are positive in qPCR; conversely, 34 membrane quarters are negative in qPCR and positive in LAMP. The presence of PCR inhibitors could explain these last results. Indeed, it has already been proven that, in contrast to qPCR, LAMP is robust against inhibitors [29, 63].

Development of field friendly amplification methods such as LAMP need to be accompanied by the development of a field-friendly pre-process of the sample. Use of DNA extraction kits that have a low yield due to their high purification level requires special apparatuses such as centrifuges, which are costly. Therefore, we have developed a new field-friendly water sample pre-process to concentrate the eDNA. The sampling and pre-process protocol were done in 45 min in the field without electricity, which represents a significant advance in SBDs monitoring. Nevertheless, several improvements can be made to our protocol, especially on the water filtration step. The manual pump used limits the speed and convenience of the protocol since it took 30 min per sample to filter just 500 ml of water with difficulties. A 12-V battery-powered diaphragm water pump coupled with an adapted filter capsule was used to filter up to 50 l water in a DNA survey targeting trematodes in water [64]. To further enhance the practicability and speed of the BteLAMP assay, an improved battery-operated pump could replace the manual pump. The compatibility between a motorised pump and the filter units used in our protocol needs to be tested.

We used the Genie III to obtain our field results from Senegal, which is a useful, easily transportable, batteryoperated field tool, allowing visualization of reaction kinetics. Coupled with freeze-dried reagents, this type of portable device makes it possible to carry out the entire protocol (sampling, pre-process, amplification and revelation) in the field [32]. We should use another portable device for low-income countries where the use of the Genie III may be a financial obstacle. The COVID-19 global pandemic involved the development of several tools to perform low-cost and field-friendly LAMPs [34, 35, 65–67]. A low-cost and open-source LAMP diagnostic device has been developed that allows realtime amplification and fluorescence reading in the same way as the Genie III [34]. It has even been proven that the LAMP reaction can be done using a coffee capsule filled with phase change material, called a “T-cup”, allowing incubation at 63 °C in boiling water and a nakedeye revelation [35]. In another study, the revelation step of a smartphone-based LAMP could be assessed with a smartphone application coupled with filters allowing the fluorescence reading. In the future, the current LAMP test should be improved with freeze-dried reagents and this type of device and then tested on a large sampling.

Conclusions

In this study, we developed a full field-friendly LAMP assay detecting the presence of the trematode intermediate snail host *B. truncatus* DNA in the environment. Due to LAMP's advantages of being field-friendly and allowing cost-effective pre-process samples, Bt-eLAMP test may be a valuable new tool for quickly and efficiently establishing recurrent risk maps for urogenital schistosomiasis. eLAMP has also been used to detect the presence of intestinal parasites in wastewater during treatment, even

offering the possibility of looking for *Schistosoma sp.* eDNA directly [68]. It would be necessary to develop eLAMP for other *Bulinus* or *Biomphalaria* species responsible for the transmission of *Schistosoma*. eLAMP could also be developed for other SBDs that have the same diagnostic pressures as schistosomiasis

Abbreviations

SBD Snail-borne disease
LAMP Loop-mediated isothermal amplification
eLAMP Environmental loop-mediated isothermal amplification
PCR Polymerase chain reaction
qPCR Quantitative polymerase chain reaction
ddPCR Droplet digital polymerase chain reaction
eDNA Environmental DNA
LOD Limit of detection
C+ Positive control
C- Negative control

Supplementary Information

The online version contains supplementary material available at <https://doi.org/10.1186/s13071-023-05705-4>.

Additional file 1.

Alignment of the Bt-eLAMP primers to the ITS2 marker (accession number: MG757890). Each arrow represents a primer. The green shades represent the direction of the primer hybridization (dark green: forward; light green: backward).

Acknowledgements

Special thanks to Alassane Ndiaye who helped collect the environmental DNA samples in the field. We thank Celia Robin who worked on the project during her internship and designed the additional LOOP primers.

Author contributions

JP, JB, OR and MB conceived the original idea and designed the study. MB performed the laboratory development of the Bt-eLAMP assay. JB, BS and MB carried out the field sampling. JB and BS provided malacological expertise. SM provided the eDNA bank. JB, JP and MB analysed and interpreted the data. JB and MB drafted the manuscript. BS, JB, SM, OR, JP and MB revised the manuscript. All authors read and approved the final manuscript.

Funding

This study was funded by the French Agency for Food, Environmental and Occupational Health & Safety (PNRES 2019/1/059 Molrisk) and the Occitanie Region (Schistodiag program). This study was carried out with the support of LabEx CeMEB, an ANR "Investissements d'avenir" program (ANR-10-LABX-04-01), and within the framework of the "Laboratoires d'Excellences (LABEX)" TULIP (ANR-10LABX-41).

Availability of data and materials

All data generated or analysed during this study are included in this published article.

Declarations

Ethics approval and consent to participate

Not applicable.

Consent for publication

Not applicable.

Competing interests

The authors declare that they have no competing interests

Table 1 Snail species used for assessing the specificity of the developed *Bulinus truncatus* LAMP primer sets

Snail species	Origin	Sampling date
<i>Bulinus truncatus</i>	Diama, Senegal	2022
	Lampsar, Senegal	2022
	Mbane, Senegal	2022
	Corsica, France	2018
	Saneinte, Senegal	2022
<i>Bulinus globosus</i>	Lampsar, Senegal	2022
<i>Bulinus senegalensis</i>	Podor, Senegal	2022
<i>Bulinus forskalli</i>	Khodit, Senegal	2022
<i>Bulinus umbilicatus</i>	Barkedji, Senegal	2022
<i>Biomphalaria glabrata</i>	Brazil	2018
<i>Bithynia tentaculata</i>	Corsica, France	2018
<i>Ancylus fluviatilis</i>	Corsica, France	2018
<i>Gyraulus laevis</i>	Corsica, France	2018
<i>Physa acuta</i>	Corsica, France	2018
<i>Potamopyrgus antipodarum</i>	Corsica, France	2018
<i>Pisidium casertanum</i>	Corsica, France	2018

Table 2 Primer sequences designed for *Bulinus truncatus* eLAMP assays

Target	Primers	Sequences (5'-3')	Sequence length (bp)	Melting temperature (°C)
Internal transcribed spacer 2 (ITS2)	F3	GTCCTTCCTGTCCCCCTT	18	59.79
	B3	CGTCACATGTCACCTCACA	20	61.59
	F2	ACGCATGTCGCCGTTAC	17	59.33
	F1c	AAGAGCACGAGCCGGGTCC	19	66.28
	B2	CGCGTTATAGCCCATCTCAA	20	59.37
	B1c	GCGAGTCATACGGACCTCGTG	21	64.21
	LB	CACAGTCTCTCCCCACACC	20	63.67
	LF	CCTGGCGACGAGATCACC	20	63.89

Table 3 Cross table comparing LAMP and qPCR analyses of *Bulinus truncatus* eDNA with kappa agreement

No. of samples				
qPCR results	LAMP positive	LAMP negative	Total	Kappa value
Positive	76	15	91	0.51
Negative	34	75	109	
Total	110	90	200	

Fig. 1 Specificity and LOD (limit of detection) of the *Bulinus truncatus* eLAMP (Bt-eLAMP) assay. Results obtained with agarose gel electrophoresis after 45 min of reaction. a Specificity; lanes 1–5, *Bulinus truncatus* DNA templates from five different localities (Diama, Lampsar, Mbane, Corsica and Saneinte); lane 6, *Bulinus globosus*; lane 7, *Bulinus senegalensis*; lane 8, *Bulinus forskalli*; lane 9, *Bulinus umbilicatus*; lane 10, *Biomphalaria glabrata*; lane 11, *Bithynia tentaculata*; lane 12, *Ancylus fluviatilis*; lane 13, *Gyraulus laevis*; lane 14, *Physa acuta*; lane 15, *Potamopyrgus antipodarum*; lane 16, *Pisidium casertanum*; lane C + , positive control (*Bulinus truncatus* DNA; 1 ng); lane C -, negative control (water as template). b LOD; lane 1–11, *Bulinus truncatus* DNA tenfold serial dilutions (10 ng/μL to 1 ag/μl); lane C -, negative control (water as template)

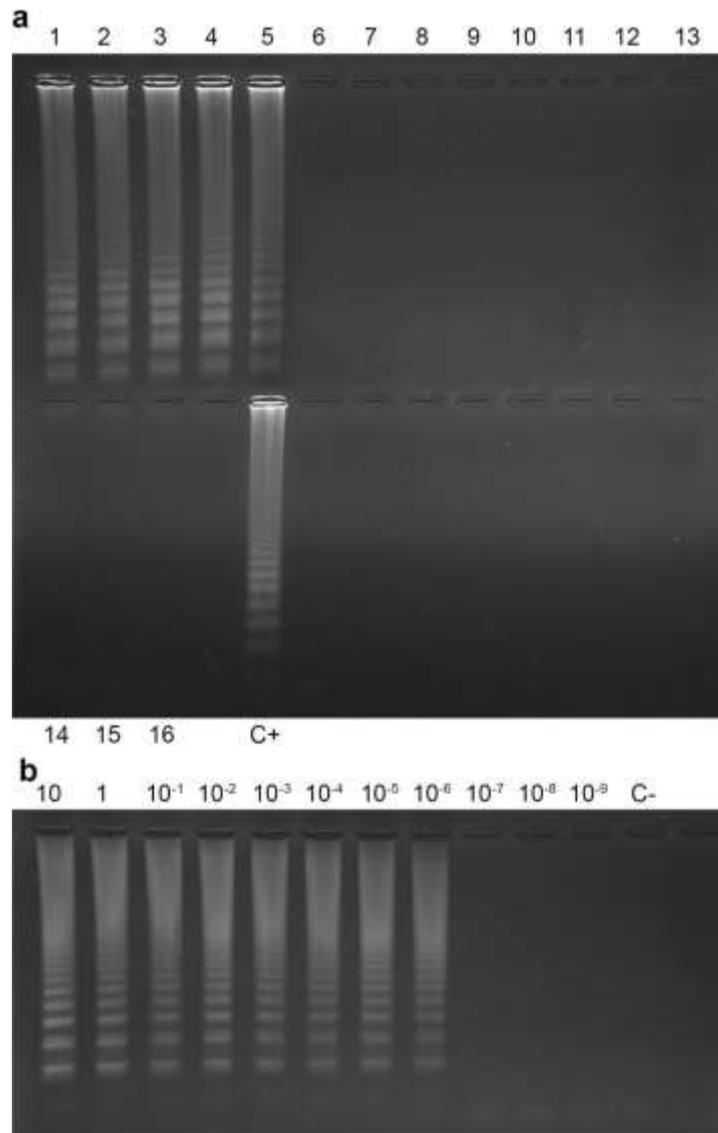
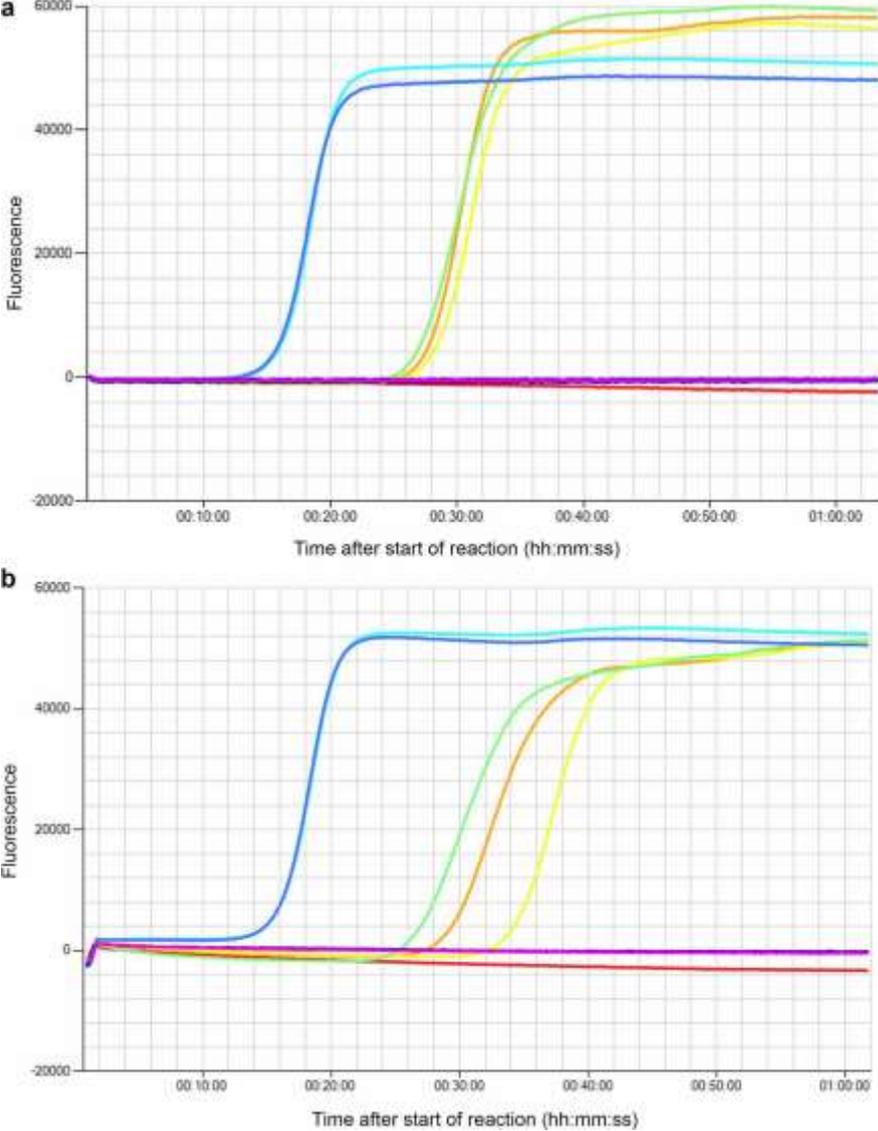


Fig. 2 Comparison of *Bulinus truncatus* environmental DNA detection by loop-mediated isothermal amplification (LAMP), quantitative polymerase chain reaction (qPCR) and droplet digital polymerase chain reaction (ddPCR) amplification methods. Each cell represents DNA amplification results from eDNA collected in two Corsican rivers (Cavu and Solenzara) and Perpignan Channel; *Bulinus truncatus* snails are present in all sites except the water intake site in the Cavu River and university site in the Perpignan Channel. Three volumes of water (1, 3 and 5 l) from both the shore and the streambeds of the rivers were collected in all Corsican sites. Each line represents the amplification result from each site using LAMP, qPCR or ddPCR amplification method. qPCR and ddPCR results come from Mulero et al. [21]. A green membrane means that at least one quarter of the filtration membrane is positive for *Bulinus truncatus* presence and red membrane means that no quarter of the filtration membrane is positive. Empty cells mean that no filtration was done under these conditions

River name	Sampling site	Detection method	1L Shore	3L Shore	5L Shore	1L Streambed	3L Streambed	5L Streambed	Control
Cavu	Water intake	LAMP	●	●	●	●	●	●	●
		qPCR	●	●	●	●	●	●	●
		ddPCR	●	●	●	●	●	●	●
	Mulinu Bridge 1	LAMP	●	●	●	●	●	●	●
		qPCR	●	●	●	●	●	●	
		ddPCR	●	●	●	●	●	●	
	Mulinu Bridge 2	LAMP	●	●	●	●	●	●	●
		qPCR	●	●	●	●	●	●	
		ddPCR	●	●	●	●	●	●	
	A Tyroliana Park 1	LAMP	●	●	●	●	●	●	●
		qPCR	●	●	●	●	●	●	
		ddPCR	●	●	●	●	●	●	
	A Tyroliana Park 2	LAMP	●	●	●	●	●	●	●
		qPCR	●	●	●	●	●	●	
		ddPCR	●	●	●	●	●	●	
3 Pool	LAMP	●	●	●	●	●	●	●	
	qPCR	●	●	●	●	●	●		
	ddPCR	●	●	●	●	●	●		
Solenzara	Solenzara Snack	LAMP	●	●	●	●	●	●	●
		qPCR	●	●	●	●	●	●	
		ddPCR	●	●	●	●	●	●	
Perpignan channel	University	LAMP	●	●					●
		qPCR	●	●					
		ddPCR	●	●					

Fig. 3 Real-time loop-mediated isothermal amplification of *Bulinus truncatus* environmental DNA in Diama (a) and Lampsar (b) sites, Senegal. Orange, yellow and green lines are tested samples. Turquoise and blue lines are positive controls (1 ng of *B. truncatus* extracted DNA). Red line is field negative sample (1500 ml filtration of mineral water). Violet and pink lines are laboratory negative samples (ultrapure water).



References

1. Nii-Trebi NI. Emerging and neglected infectious diseases: insights, advances, and challenges. *Biomed Res Int*. 2017;2017:5245021.
2. Baker RE, Mahmud AS, Miller IF, Rajeev M, Rasambainarivo F, Rice BL, et al. Infectious disease in an era of global change. *Nat Rev Microbiol*. 2022;20:193–205.
3. Tredennick AT, O’Dea EB, Ferrari MJ, Park AW, Rohani P, Drake JM. Anticipating infectious disease re-emergence and elimination: a test of early warning signals using empirically based models. *J R Soc Interface*. 2022;19:20220123.
4. Caminade C, McIntyre KM, Jones AE. Impact of recent and future climate change on vector-borne diseases. *Ann NY Acad Sci*. 2019;1436:157–73.
5. Allen T, Murray KA, Zambrana-Torrel C, Morse SS, Rondinini C, Di Marco M, et al. Global hotspots and correlates of emerging zoonotic diseases. *Nat Commun*. 2017;8:1124.
6. Gonzalez G, Bournez L, Moraes RA, Marine D, Galon C, Vorimore F, et al. A one-health approach to investigating an outbreak of alimentary tickborne encephalitis in a non-endemic area in France (Ain, Eastern France): a longitudinal serological study in livestock, detection in ticks, and the first tick-borne encephalitis virus isolation and molecular characterisation. *Front Microbiol*. 2022;13:863725.
7. Salas-Coronas J, BARGUES MD, Lozano-Serrano AB, Artigas P, Martinez-Orti A, Mas-Coma S, et al. Evidence of autochthonous transmission of urinary schistosomiasis in Almeria (southeast Spain): an outbreak analysis. *Travel Med Infect Dis*. 2021;44:102165.
8. Boissier J, Mone H, Mitta G, BARGUES MD, Molyneux D, Mas-Coma S. Schistosomiasis reaches Europe. *Lancet Infect Dis*. 2015;15:757–8.
9. Boissier J, Grech-Angelini S, Webster BL, Allienne J-F, Huyse T, Mas-Coma S, et al. Outbreak of urogenital schistosomiasis in Corsica (France): an epidemiological case study. *Lancet Infect Dis*. 2016;16:971–9.
10. Noel H, Ruello M, Maccary A, Pelat C, Sommen C, Boissier J, et al. Large outbreak of urogenital schistosomiasis acquired in Southern Corsica, France: monitoring early signs of endemicization? *Clin Microbiol Infect*. 2018;24:295–300.
11. Berry A, Fillaux J, Martin-Blondel G, Boissier J, Iriart X, Marchou B, et al. Evidence for a permanent presence of schistosomiasis in Corsica, France, 2015. *Euro Surveill*. 2016. <https://doi.org/10.2807/1560-7917.ES.2016.21.1.30100>.
12. El-Sayed A, Kamel M. Climatic changes and their role in emergence and re-emergence of diseases. *Environ Sci Pollut Res Int*. 2020;27:22336–52.
13. Giron S, Fournet N, Franke F, Brottet E, Calba C, Cochet A, et al. Review of Arbovirus surveillance in 2019: transition to surveillance for confirmed cases of Chikungunya, Dengue and Zika Virus in metropolitan France. *BEH*. 2019;22:446–55.
14. Musso D, Gubler DJ. Zika virus. *Clin Microbiol Rev*. 2016;29:487–524.
15. Myrsterud A, Easterday WR, Stigum VM, Aas AB, Meisingset EL, Viljugrein H. Contrasting emergence of Lyme disease across ecosystems. *Nat Commun*. 2016;7:11882.
16. Millins C, Leo W, MacInnes I, Ferguson J, Charlesworth G, Nayar D, et al. Emergence of Lyme disease on Treeless Islands, Scotland, United Kingdom. *Emerg Infect Dis*. 2021;27:538–46.
17. Kraemer MUG, Hay SI, Pigott DM, Smith DL, Wint GRW, Golding N. Progress and challenges in infectious disease cartography. *Trends Parasitol*. 2016;32:19–29.
18. Rollinson D, Knopp S, Levitz S, Stothard JR, Tchuem Tchuente L-A, Garba A, et al. Time to set the agenda for schistosomiasis elimination. *Acta Trop*. 2013;128:423–40.
- 19 King CH, Sutherland LJ, Bertsch D. Systematic review and meta-analysis of the impact of chemical-based Mollusciciding for control of *Schistosoma mansoni* and *S. haematobium* transmission. *PLOS Negl Trop Dis*. 2015;9:e0004290.
20. Sokolow SH, Wood CL, Jones IJ, Swartz SJ, Lopez M, Hsieh MH, et al. Global assessment of schistosomiasis control over the past century shows targeting the snail intermediate host works best. *PLoS Negl Trop Dis*. 2016;10:e0004794.

21. Mulero S, Boissier J, Allienne J-F, Quilichini Y, Foata J, Pointier J-P, et al. Environmental DNA for detecting *Bulinus truncatus*: a new environmental surveillance tool for schistosomiasis emergence risk assessment. *Environ DNA*. 2020;2:161–74.
22. Lamy T, Pointier J, Jarne P, David P. Testing metapopulation dynamics using genetic, demographic and ecological data. *Mol Ecol*. 2012;21:1394–410.
23. Kamel B, Laidemitt MR, Lu L, Babbitt C, Weinbaum OL, Mkoji GM, et al. Detecting and identifying *Schistosoma* infections in snails and aquatic habitats: a systematic review. *PLoS Negl Trop Dis*. 2021;15:e0009175.
24. Schneider J, Valentini A, Dejean T, Montarsi F, Taberlet P, Glaizot O, et al. Detection of invasive mosquito vectors using environmental DNA (eDNA) from water samples. *PLoS ONE*. 2016;11:e0162493.
25. Jones RA, Brophy PM, Davis CN, Davies TE, Emberson H, Rees Stevens P, et al. Detection of *Galba truncatula*, *Fasciola hepatica* and *Calicophoron daubneyi* environmental DNA within water sources on pasture land, a future tool for fluke control? *Parasit Vectors*. 2018;11:342.
26. Rathinasamy V, Hosking C, Tran L, Kelley J, Williamson G, Swan J, et al. Development of a multiplex quantitative PCR assay for detection and quantification of DNA from *Fasciola hepatica* and the intermediate snail host, *Austropeplea tomentosa*, in water samples. *Vet Parasitol*. 2018;259:17.
27. Calata FIC, Caranguian CZ, Mendoza JEM, Fornillos RJC, Tabios IKB, Fontanilla IKC, et al. Analysis of environmental DNA and edaphic factors for the detection of the snail intermediate host *Oncomelania hupensis quadrasi*. *Pathogens*. 2019;8:160.
28. Kincaid-Smith J, Rey O, Toulza E, Berry A, Boissier J. Emerging Schistosomiasis in Europe: a need to quantify the risks. *Trends Parasitol*. 2017;33:600–9.
29. Mori Y, Nagamine K, Tomita N, Notomi T. Detection of loop-mediated isothermal amplification reaction by turbidity derived from magnesium pyrophosphate formation. *Biochem Biophys Res Commun*. 2001;289:150–4.
30. Notomi T, Okayama H, Masubuchi H, Yonekawa T, Watanabe K, Amino N, et al. Loop-mediated isothermal amplification of DNA. *Nucleic Acids Res*. 2000;28:E63.
31. Notomi T, Mori Y, Tomita N, Kanda H. Loop-mediated isothermal amplification (LAMP): principle, features, and future prospects. *J Microbiol*. 2015;53:1–5.
32. Kumar S, Sharma S, Bhardwaj N, Pande V, Savargaonkar D, Anvikar AR. Advanced lyophilised loop mediated isothermal amplification (L-LAMP) based point of care technique for the detection of dengue virus. *J Virol Methods*. 2021;293:114168.
33. Kumar Y. Isothermal amplification-based methods for assessment of microbiological safety and authenticity of meat and meat products. *Food Control*. 2021;121:107679.
34. Buultjens AH, Vandellanoot K, Sharkey LK, Howden BP, Monk IR, Lee JYH, et al. Low-cost, open-source device for high-performance fluorescence detection of isothermal nucleic acid amplification reactions. *ACS Biomater Sci Eng*. 2021;7:4982–90.
35. Velders AH, Ossendrijver M, Keijser BJF, Saggiomo V. T-cup: a cheap, rapid, and simple home device for isothermal nucleic acid amplification. *Glob Chall*. 2022;6:2100078.
36. Chen Z, Yang T, Yang H, Li T, Nie L, Mou X, et al. A portable multi-channel turbidity system for rapid detection of pathogens by loop-mediated isothermal amplification. *J Biomed Nanotechnol*. 2018;14:198–205.
37. Matute T, Nunez I, Rivera M, Reyes J, Blazquez-Sanchez P, Arce A, et al. Homebrew reagents for low cost RT-LAMP. *J Biomol Tech*. 2021. <https://doi.org/10.7171/jbt.21-3203-006>.
38. Detemmerman L, Olivier S, Bours V, Boemer F. Innovative PCR without DNA extraction for African sickle cell disease diagnosis. *Hematology*. 2018;23:181–6.
39. Gandasegui J, Fernandez-Soto P, Carranza-Rodriguez C, Perez-Arellano JL, Vicente B, Lopez-Aban J, et al. The rapid-heat LAMPellet method: a potential diagnostic method for human urogenital Schistosomiasis. *PLoS Negl Trop Dis*. 2015;9:e0003963.
40. Lee S, Khoo VSL, Medriano CAD, Lee T, Park S-Y, Bae S. Rapid and in-situ detection of fecal indicator bacteria in water using simple DNA extraction and portable loop-mediated isothermal amplification (LAMP) PCR methods. *Water Res*. 2019;160:371–9.

41. Davis CN, Tyson F, Cutress D, Davies E, Jones DL, Brophy PM, et al. Rapid detection of *Galba truncatula* in water sources on pasture-land using loop-mediated isothermal amplification for control of trematode infections. *Parasit Vectors*. 2020;13:496.
42. Williams MR, Stedtfeld RD, Engle C, Salach P, Fakher U, Stedtfeld T, et al. Isothermal amplification of environmental DNA (eDNA) for direct fieldbased monitoring and laboratory confirmation of *Dreissena* sp. *PLoS ONE*. 2017;12:e0186462.
43. Feist SM, Lance RF. Genetic detection of freshwater harmful algal blooms: a review focused on the use of environmental DNA (eDNA) in *Microcystis aeruginosa* and *Prymnesium parvum*. *Harmful Algae*. 2021;110:102124.
44. World Health Organization Expert Committee on the Control of Schistosomiasis. Prevention and control of schistosomiasis and soil-transmitted helminthiasis : report of a WHO expert committee. Geneve. 2002. <https://apps.who.int/iris/handle/10665/42588>. Accessed 19 Apr 2022.
45. Stensgaard A-S, Vounatsou P, Sengupta ME, Utzinger J. Schistosomes, snails and climate change: current trends and future expectations. *Acta Trop*. 2019;190:257–68.
46. Seventieth World Health Assembly. Global vector control response: an integrated approach for the control of vector-borne diseases. Report No.:WHA70.16. Geneve. 2017. [https:// apps. who. int/ iris/ handle/ 10665/ 275708](https://apps.who.int/iris/handle/10665/275708). Accessed 19 Apr 2022.
47. Berry A, Mone H, Iriart X, Mouahid G, Aboo O, Boissier J, et al. *Schistosomiasis haematobium*, Corsica. *France Emerg Infect Dis*. 2014;20:1595–7.
48. Zein-Eddine R, Djuikwo-Teukeng FF, Al-Jawhari M, Senghor B, Huyse T, Dreyfuss G. Phylogeny of seven *Bulinus* species originating from endemic areas in three African countries, in relation to the human blood fluke *Schistosoma haematobium*. *BMC Evol Biol*. 2014;14:271.
49. Chibwana FD, Tumwebaze I, Mahulu A, Sands AF, Albrecht C. Assessing the diversity and distribution of potential intermediate hosts snails for urogenital schistosomiasis: *Bulinus* spp. (Gastropoda: Planorbidae) of Lake Victoria. *Parasites Vectors*. 2020;13:418.
50. Abe EM, Guo Y-H, Shen H, Mutsaka-Makuvaza MJ, Habib MR, Xue J-B, et al. Phylogeography of *Bulinus truncatus* (Audouin, 1827) (Gastropoda: Planorbidae) in selected African Countries. *Trop Med Infect Dis*. 2018;3:127.
51. Sochorova J, Garcia S, Galvez F, Symonova R, Kovařík A. Evolutionary trends in animal ribosomal DNA loci: introduction to a new online database. *Chromosoma*. 2018;127:141–50.
52. Gregory TR, Nicol J, Tamm H, Kullman B, Kullman K, Leitch I, et al. Eukaryotic genome size database. *Nucleic Acids Res*. 2007;35:D332–8.
53. Prokopowich CD, Gregory TR, Crease TJ. The correlation between rDNA copy number and genome size in eukaryotes. *Genome*. 2003;46:48–50.
54. Iwanaga T, Anzawa K, Mochizuki T. Variations in ribosomal DNA copy numbers in a genome of *Trichophyton interdigitale*. *Mycoses*. 2020;63:1133–40.
55. Malone JH. Balancing copy number in ribosomal DNA. *Proc Natl Acad Sci USA*. 2015;112:2635–6.
56. Johnson SM, Carlson EL, Pappagianis D. Determination of ribosomal DNA copy number and comparison among strains of *Coccidioides*. *Mycopathologia*. 2015;179:45–51.
57. Abbasi I, King CH, Muchiri EM, Hamburger J. Detection of *Schistosoma mansoni* and *Schistosoma haematobium* DNA by Loop-mediated isothermal amplification: identification of infected snails from early prepatency. *Am J Trop Med Hyg*. 2010;83:427–32.
58. Gandasegui J, Fernandez-Soto P, Hernandez-Goenaga J, Lopez-Aban J, Vicente B, Muro A. Biomphal-LAMP: a new rapid loop-mediated isothermal amplification assay for detecting *Schistosoma mansoni* in *Biomphalaria glabrata* snail host. *PLoS Negl Trop Dis*. 2016;10:e0005225.
59. Caldeira RL, Jannotti-Passos LK, Jannotti-Passos LK, Dos Santos Carvalho O. Use of molecular methods for the rapid mass detection of *Schistosoma mansoni* (Platyhelminthes: Trematoda) in *Biomphalaria* spp. (Gastropoda: Planorbidae). *J Trop Med*. 2017;2017:8628971.
60. Falzone L, Musso N, Gattuso G, Bongiorno D, Palermo CI, Scalia G, et al. Sensitivity assessment of droplet digital PCR for SARS-CoV-2 detection. *Int J Mol Med*. 2020;46:957–64.

61. Kojabad AA, Farzanehpour M, Galeh HEG, Dorostkar R, Jafarpour A, Bolandian M, et al. Droplet digital PCR of viral DNA/RNA, current progress, challenges, and future perspectives. *J Med Virol*. 2021;93:4182–97.
62. Catalano S, Leger E, Fall CB, Borlase A, Diop SD, Berger D, et al. Multihost transmission of *Schistosoma mansoni* in Senegal, 2015–2018. *Emerg Infect Dis*. 2020;26:1234–42.
63. Kaneko H, Kawana T, Fukushima E, Suzutani T. Tolerance of loop-mediated isothermal amplification to a culture medium and biological substances. *J Biochem Biophys Methods*. 2007;70:499–501.
64. Douchet P, Boissier J, Mulero S, Ferte H, Doberva M, Allienne J-F, et al. Make visible the invisible: optimized development of an environmental DNA metabarcoding tool for the characterization of trematode parasitic communities. *Environ DNA*. 2022;4:627–41.
65. Rezaei M, Razavi Bazaz S, Morshedi Rad D, Shimoni O, Jin D, Rawlinson W, et al. A portable RT-LAMP/CRISPR machine for rapid COVID-19 screening. *Biosensors (Basel)*. 2021;11:369.
66. Sreejith KR, Umer M, Dirr L, Bailly B, Guillon P, von Itzstein M, et al. A portable device for LAMP based detection of SARS-CoV-2. *Micromachines (Basel)*. 2021;12:1151.
67. Choi G, Guan W. An ultracompact real-time fluorescence loopmediated isothermal amplification (LAMP) analyzer. *Methods Mol Biol*. 2022;2393:257–78.
68. Ajonina C, Buzie C, Moller J, Otterpohl R. The detection of *Entamoeba histolytica* and *Toxoplasma gondii* in wastewater. *J Toxicol Environ Health Part A*. 2018;81:1–5.