Since January 2020 Elsevier has created a COVID-19 resource centre with free information in English and Mandarin on the novel coronavirus COVID-19. The COVID-19 resource centre is hosted on Elsevier Connect, the company's public news and information website.

occurred. When a MALDI result of *M. abscessus* subsp. *massiliense* with a high score is achieved, the isolate may be confidently identified to subspecies level, which is highly predictive of macrolide susceptibility.[3]

Prior to the MALDI-TOF era, some organisms were proven to be difficult to identify in routine laboratories. The usage of proteomics-based mass spectrometry has revolutionised operations and MALDI is now commonly used for identifying bacteria, fungi, and mycobacteria due to rapidity as well as accuracy. In the context of mycobacteriology, MALDI-TOF can identify commonly encountered NTM species, but is currently unable to identify closely related organisms such as *M. abscessus* to subspecies level. This is consistent with previous reports.[8,9] However, we note that a MALDI result of *M. abscessus* subsp. *massiliense* is relatively specific, particularly with scores of $\geq$2.100. Such a result would be highly predictive of the identification being clarithromycin-susceptible *M. abscessus* subsp. *massiliense,* and may be useful in tailoring empiric antimicrobial therapy particularly for critically ill or immunocompromised patients. Further improvements to the current proteomics-based libraries are required before more reliable subspecies identification can be performed using MALDI-TOF. Modifications to the MALDI approach include lipid-based mass spectrometry.[10] Initial testing suggests that generated mass spectra can differentiate the *M. abscessus* subspecies. Although potentially promising, there remains significant validation to be performed with a larger collection of isolates to ensure reproducibility across different laboratories, and this needs to be followed by development of a publicly available database.

Further advances are required for existing MALDI-TOF based technologies before confident identifications to subspecies level can be made. Currently, molecular testing methods remain the best alternative for earlier subspecies identification. This includes polymerase chain reaction testing to identify the presence of truncated or normal length *erm* (41) genes, PCR and sequencing key genes, or using commercial molecular identification methods.

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Cadaveric donor specimens and serological testing for SARS-CoV-2

To the Editor,
The coronavirus disease 2019 (COVID-19) pandemic has raised questions around the potential risk of severe acute respiratory syndrome coronavirus 2 (SARS-CoV-2) in tissue and solid organ transplant (SOT) donors and recipients. Cadavers are the major source of donor organs and tissues worldwide. There is a higher risk of severe COVID-19 outcomes related to chronic immunosuppression and associated morbidities. Testing of cadavers for SARS-CoV-2 is limited by the availability and validity of upper respiratory samples, and alternative testing is needed. Screening for SARS-CoV-2 antibodies in donor blood should be given due consideration as a simple and effective way of assessing SARS-CoV-2 infections in cadavers.[1] Clinical outcomes of tissue and organ recipients from cadaveric donors with prior SARS-CoV-2 infection remain uncertain. Approximately 70% of organs and tissues used in transplantation are obtained from deceased donors. All of these require screening for blood borne viruses. More recently with the COVID-19 pandemic, issues have arisen around reductions in donors during the pandemic and during times of increased case numbers, and how to deal with the issues of respiratory virus infection in a laboratory setting mainly directed at blood borne pathogens. Retrieval of SOT and tissues from cadavers introduces unique challenges in testing for infectious disease markers, particularly in cases of tissue donation where cadaveric blood is the only source for testing.[2] Therefore, adequate testing for SARS-CoV-2 antibodies in cadaveric blood is necessary in order to ensure the suitability and safety of the transplantation.

Existing data for evaluating serological tests for assessment of SARS-CoV-2 exposure are from living, acutely infected or recovered patients.[3,4] Fast tracked molecular biomarkers of SARS-CoV-2 infections include serological assays screening for antigens and antibodies. Commercially available serological assays are only licensed for sera collected up to 21.5 h post-mortem.[8] The Architect chemiluminescent microparticle immunoassay (CMIA) on the Abbott Architect Systems analyser (Abbott Diagnostics, Australia) assesses SARS-CoV-2 immunoglobulin class G (IgG) qualitatively and quantitatively, although there has been no evaluation on cadaveric specimens. We previously showed the utility of serological testing on this platform for other targets,[5] with HIV antigen/antibody and HCV antibody tests unaffected in post-mortem sera collected within 24 h following death.[5] Previous studies demonstrated increased incidence of false-positive serological results in post-mortem bloods, though the specificity of SARS-CoV-2 CMIA testing for cadaveric specimens is unknown.[6–8] This verification study evaluated the suitability of serum and plasma specimens collected up to 24 h post-mortem, for use in screening donors for SARS-CoV-2 antibodies. Analytical performance of the SARS-CoV-2 IgG qualitative assay compared cadaveric and living donor specimens tested using the Abbott Architect analyser.

Cadaveric (25 test specimen) and living (25 control specimen) healthy donor sera, and plasma from EDTA anticoagulated blood, were obtained from the NSW Bone Bank and Lions NSW Eye Bank. Specimens were retrospectively tested on the Abbott Architect *i*2000SR immunoassay analyser (Abbott Laboratories, USA). The duration from time of death to time of collection of these cadaveric specimens ranged between 5 h and 23 h 56 min. Only donors whose specimens were non-reactive [signal cut-off (S/CO) <1.4 index] for SARS-CoV-2 IgG by serological testing were included. These specimens were spiked with a pooled stock of reactive SARS-CoV-2 IgG specimens (S/CO ≥1.4) obtained from Serology and Virology Division (SAViD), New South Wales Health Pathology Randwick. In brief, reactive source sera used to spike the non-reactive cadaveric and living donor specimens were diluted with normal human plasma (NHP) and added to the specimens to yield reactivity near the assay signal cut-off (low level reactivity, S/CO 2–4) and high level reactivity (S/CO 6–8).

Parameters of specificity, sensitivity, accuracy, reproducibility and impact of storage conditions were assessed to compare the suitability of the Abbott Architect SARS-CoV-2 IgG assay for testing cadaveric specimens for past SARS-CoV-2 infection. Testing of all specimens was conducted using a single lot number of the SARS-CoV-2 Reagent Kit, one lot of the SARS-CoV-2 IgG Calibrator Kit, and one lot of the SARS-CoV-2 Control Kit all on the same Architect *i*2000SR analyser.

For assay specificity, a total of 25 cadaveric and living donor specimens were tested un-spiked in single replicate. The cadaveric and living donor specimens were then spiked with positive sera at S/CO ratio of 2–4 and tested in single replicate, to test the sensitivity of the assay. The accuracy of the assay was tested by the division of 20 cadaveric and 20 living donor specimens into three aliquots. The first aliquot was tested un-spiked, the second aliquot was spiked at S/CO ratio of 2–4, and the third aliquot was spiked at S/CO ratio of 6–8. The assay reproducibility was evaluated with 20 cadaveric and 20 living donor specimens spiked at S/CO ratio of 2–4 and tested in six different runs using one reagent kit lot on the same analyser. The impact of storage conditions was evaluated in 20 cadaveric specimens of 10 non-spiked and 10 spiked at S/CO ratio of 2–4. The variation of S/CO values was assessed following 72 h storage at room temperature (20–24°C), and after 144 h storage at 2–8°C.

The mean and standard deviation of the specimen S/CO were calculated for both test and control specimens. A two-tailed Student's *t* test was used to calculate significance. A *p* value of <0.05 was regarded as statistically significant. For reproducibility, the total percentage coefficient of variance (%CV) was calculated for both test and control specimens.

The average time of death to the time of collection for post-mortem blood was 16 h and 1 min, and the median was 15 h and 4 mins. The sensitivity [95% confidence interval (CI) 2.26–2.35] and specificity (95% CI 0.04–0.12) of the assay for detecting SARS-CoV-2 antibody was 100% for the test and control specimens. The *p* values for the aforementioned parameters were 0.14 and 0.79, respectively (Table 1). That is, there was no statistically significant difference between test and control specimens, which were within specified assay range. Similarly, the accuracy of the cadaveric and living donor specimens for non-spiked (95% CI 0.04–0.16), low reactive (95% CI 3.97–4.03), and high reactive (95% CI 6.31–6.39) SARS-CoV-2 IgG had *p* values between 0.07 and 0.99 and were not statistically significant (Table 1). Reproducibility was assessed by measuring the %CV of low level reactive 20 cadaveric and 20 living donor specimens tested on six separate days. The average seropositive (S/CO)

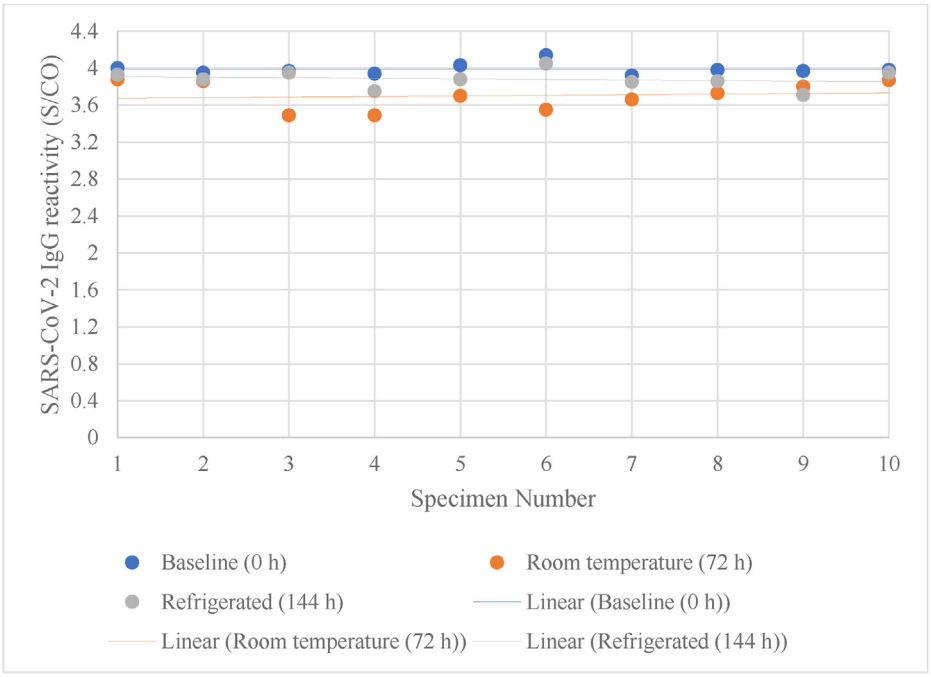
Table 1 Specificity, sensitivity, accuracy and reproducibility of the Abbott Architect SARS-CoV-2 IgG assay on living and cadaveric plasma and sera

Assay parameters	No. Samples tested	Living		Cadaveric		95% CI	*p* value	%CV
		Mean (S/CO)	SD	Mean (S/CO)	SD			
Specificity	25	0.11	0.20	0.05	0.04	0.04–0.12	0.14	–
Sensitivity	25	2.32	0.14	2.30	0.19	2.26–2.35	0.79	–
Accuracy (non-spiked)	20	0.10	0.17	0.10	0.22	0.04–0.16	0.99	–
Accuracy (low level)	20	4.03	0.09	3.97	0.08	3.97–4.03	0.07	–
Accuracy (high level)	20	6.38	0.13	6.32	0.13	6.31–6.39	0.13	–
Reproducibility	20	2.27	0.10	2.33	0.05	–	–	3.46

95% CI, 95% confidence interval; %CV, percentage coefficient of variance; –, no data; SD, standard deviation; S/CO, signal cut-off index.
The two-tailed Student's *t* test was used to determine *p* value; $p<0.05$ was statistically significant.

reactive test and control specimens was 2.33 and 2.27, respectively (Table 1). The total inter-assay %CV was 3.46, and the %CV of cadaveric specimens was comparable to specimens from healthy living donors (Table 1).

The influence of specimen storage conditions was evaluated by the signal variance on a random selection of 20 cadaveric specimens. The study tested 10 unspiked specimens and 10 spiked specimens at low level reactivity (S/CO 2−4). Each specimen was tested in singular at baseline, after 72 h storage at room temperature (20−24°C), and after 144 h storage at 2−8°C time points (Fig. 1). Each of the three time points showed linearity upon comparison of line of best fit, with no significant shift in antibody detection. These results indicated stability for antibody in both unspiked and spiked specimens.



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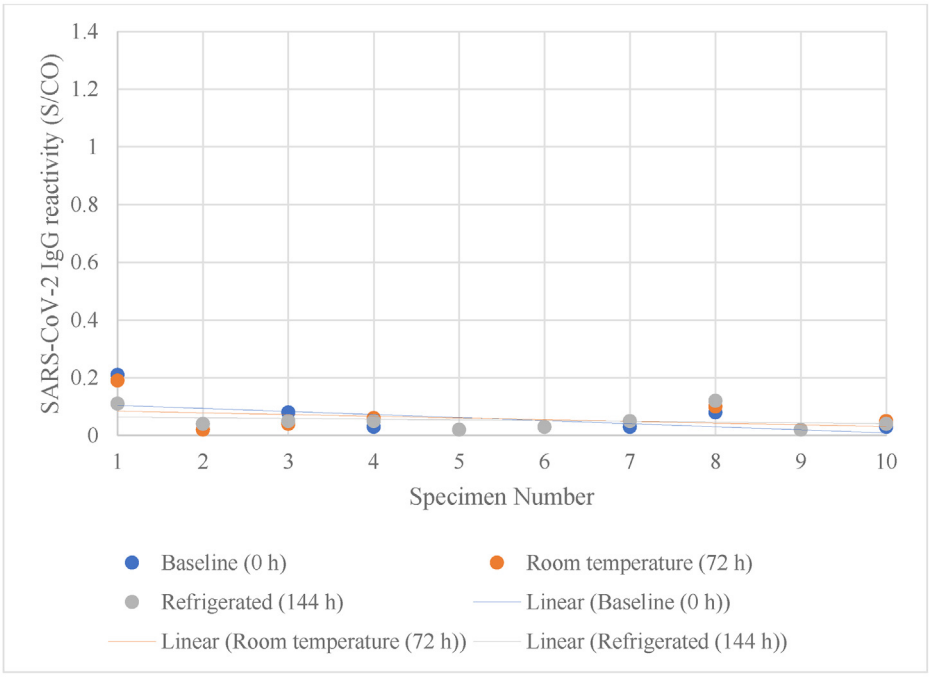

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Fig. 1 Cadaveric specimens stored at room temperature (20−24°C) for 72 h and refrigerated (2−8°C) for 144 h. (A) Signal cut-off and comparison of three timepoints on SARS-CoV-2 IgG spiked specimens. (B) Signal cut-off and comparison of three storage timepoints on non-SARS-CoV-2 reactive specimens.

Data used to assess risk of donor derived SARS-CoV-2 infection remain undefined, although case reports are emerging. In Australia, overall organ transplantation activity observed a reduction, with kidney transplantation rates down 27% compared with in 2019.[9] Internationally, the reduction of overall organ transplantation rates was more than 50% in France, Spain and the Unites States by April 2020.[10] A proven donor-to-recipient transmission of SARS-CoV-2 in a lung transplant recipient, despite negative clinical and laboratory screening of the donor, highlights the importance of additional test methods for screening.[11]

Changes in the biochemical properties of blood following death are known to adversely affect the outcomes of serological tests. Inhibitory factors observed in cadaveric specimens include an increase in free haemoglobin, potentially compromising the sensitivity and specificity of an immunoassay.[2,5] Furthermore, the performance of the Architect SARS-CoV-2 IgG assay has yet to be officially established for the use of cadaveric specimens or other specimens besides human serum or plasma by Abbott. Accurate detection of infectious markers in donors, especially cadavers, provides assurance that the presence of SARS-CoV-2 is effectively assessed and may assist the cohesive decision for tissue and SOT.

The current study demonstrated no significant difference between testing of sera from living and cadaveric individuals for the examined parameters for SARS-CoV-2 IgG. Furthermore, the storage study support claims established for cadaveric specimens, where it showed no significant shift of up to 72 h at 20−24°C and up to 144 h at 2−8°C. This indicates testing of human serum and plasma specimens collected up to 24 h post-mortem with the Abbott Architect SARS-CoV-2 IgG assay is acceptable.

The consistency between the test and control specimens for detection of past infection with SARS-CoV-2 using specific antibody detection, reflects assay capability for use in donor screening. Furthermore, detection of COVID-19 antibodies in conjunction with SARS-CoV-2 RT-PCR may alleviate the hesitancy surrounding donor derived infections. This could contribute to reducing the burden of COVID-19 on declining transplantation rates and increase the availability of potentially lifesaving organs.[12]

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Analysis of SARS-CoV-2 real-time PCR test CT values across a population may afford useful information to assist public health efforts and add refinement to epidemiological models

To the Editor,
Initial public health measures were effective at reducing transmission of SARS-CoV-2 in Australia. In New South Wales in late December 2021, relaxation of these measures coupled with a seasonal change in people movement and the emergence of the Omicron strain (B.1.1.529) led to a surge in community transmission. During the peak period in January, demand for polymerase chain reaction (PCR) testing exceeded capacity, and the true community prevalence of infection was unknown and likely underestimated. A decline in case