


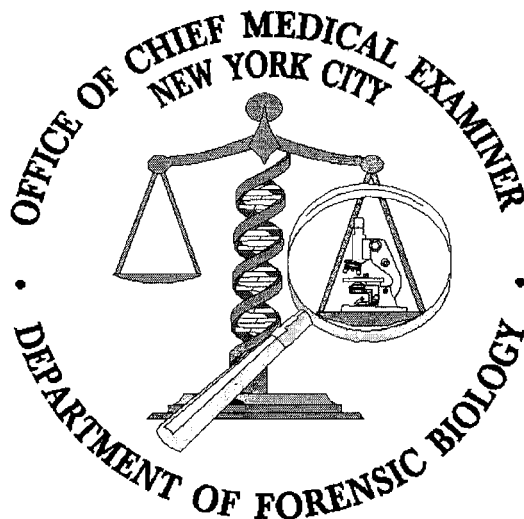


REVISION

PROTOCOLS FOR FORENSIC STR TYPING (SECTION 2 – DNA EXTRACTION)

Effective date: May 26, 2006

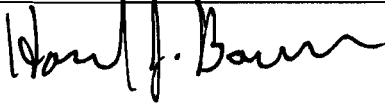
REVIEWED/APPROVED BY			
Title	Print Name	Signature	Date
Deputy Director / Technical Leader	Howard J. Baum, PhD		May 26, 2006

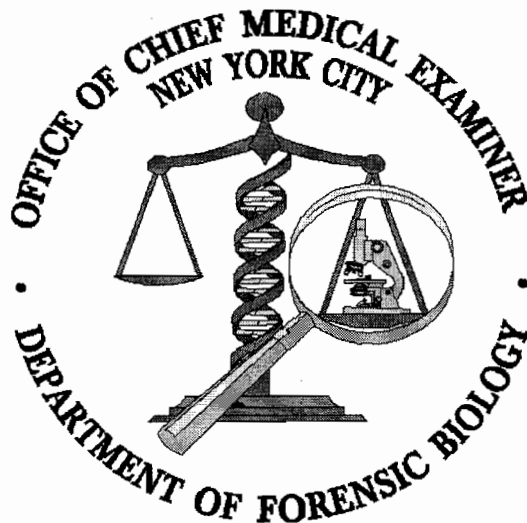


REVISION

PROTOCOLS FOR FORENSIC STR TYPING (SECTION 3B)

Effective date: June 1, 2007

REVIEWED/APPROVED BY			
Title	Print Name	Signature	Date
Deputy Director / Technical Leader	Howard J. Baum, Ph.D.		June 1, 2007



REVISION

PROTOCOLS FOR FORENSIC STR TYPING [SECTION 3 (Section 3A and Section 3B)]

Effective date: October 5, 2006

REVIEWED/APPROVED BY			
Title	Print Name	Signature	Date
Deputy Director / Technical Leader	Howard J. Baum, Ph.D.	<i>Howard J. Baum</i>	October 5, 2006

FORENSIC BIOLOGY PROTOCOLS FOR FORENSIC STR ANALYSIS

1. GENERAL GUIDELINES FOR DNA CASEWORK		
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Body fluid identification

1. The general laboratory policy is to identify the stain type, i.e., usually blood, semen, or saliva (see the Biochemistry Manual) before individualization is attempted. However, circumstances will exist when this may not be possible.
2. A positive and interpretable QuantiBlot and/or STR result can be considered human (primate) positive. Identification of the specific physiological fluid may be accomplished using the procedures described in the Biochemistry Manual. A positive screening test for blood followed by the detection of a positive QuantiBlot is reported as "human blood was found".

Laboratory organization

1. To minimize the potential for carry-over contamination, the laboratory is organized so that the areas for DNA extraction, for PCR set-up, and for handling amplified DNA are physically isolated from each other. Each of the three areas is in a separate room.
2. Each sample handling area should have its own microfuge racks. The racks should only leave their designated area to transport samples to the next area. Immediately after transporting samples, the racks should be returned to their designated area.
3. Dedicated equipment such as pipetters should not leave their designated areas. Only the samples in designated racks should move between areas.

Microfuge tube and pipet handling

1. Avoid splashes and aerosols. Centrifuge all liquid to the bottom of a closed microfuge tube before opening it.
2. Avoid touching the inside surface of the tube caps with pipettes, gloves, or lab coat sleeves.

Use the correct pipette for the volume to be pipetted. Generally, the range of a pipette begins at 10% of its maximum volume (i.e., a 100 uL pipette can be used for volumes of 10 - 100 uL).
3. Use filter pipette tips for pipetting all DNA and use whenever possible for other reagents. Use the appropriate filter tips for the different sized pipettes; the tip of the pipette should never touch the filter.

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4. Always change pipet tips between handling each sample.
5. Never "blow out" the last bit of sample from a pipet. Blowing out increases the potential for aerosols, which may contaminate a sample with DNA from other samples. The accuracy of liquid volume delivered is not critical enough to justify blowing out.
6. Discard pipet tips if they accidentally touch the bench paper or any other surface.
7. Wipe the outside of the pipette with 10% bleach solution if the barrel goes inside a tube.

Sample handling

1. Samples that have not yet been amplified should never come in contact with equipment in the amplified DNA work area. Samples that have been amplified should never come in contact with equipment in the unamplified work area.
2. The DNA extraction and PCR setup of evidence samples should be performed at a separate time from the DNA extraction and PCR setup of exemplars. This precaution will help to prevent potential cross-contamination between evidence samples and exemplars.
3. Use disposable bench paper to prevent the accumulation of human DNA on permanent work surfaces. 10% bleach should be used periodically to decontaminate exposed work surfaces.
4. Limit the quantity of samples handled in a single run to a manageable number. This precaution will reduce the risk of sample mix-up and the potential for sample-to-sample contamination.
5. Change gloves frequently to avoid sample-to-sample contamination. Change them whenever they might have been contaminated with DNA and whenever exiting a sample handling area.
6. Make sure worksheets and logbooks are completely filled out.

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DNA Extraction Guidelines

Slightly different extraction procedures may be required for each type of specimen. Due to the varied nature of evidence samples, the user may need to modify procedures.

1. All tube set-ups must be witnessed **prior** to starting the extraction (**NOTE:** For differential extractions, the tube set-up should be witnessed after the incubation step.)
2. Use Kimwipes or a tube opener to open sample tubes; Only one tube should be uncapped at a time.
3. When pouring or pipetting Chelex solutions, the resin beads must be distributed evenly in solution. This can be achieved by shaking or vortexing the tubes containing the Chelex stock solution before aliquoting.
4. For pipetting Chelex, the pipette tip used must have a relatively large bore – 1 mL pipette tips are adequate.
5. Be aware of small particles of fabric, which may cling to the outside of tubes.
6. Include an extraction negative control with each batch of extractions to demonstrate extraction integrity. The extraction negative control contains water in place of biological fluids or stains. If DNA is found in the extraction negative control by QuantiBlot analysis, the extraction of all the samples in the batch should be repeated and the samples should not be amplified. However, if no DNA is found then the extraction negative controls are treated as normal samples and amplified and typed along with the test samples.
7. If a sample is found to contain <0.15 ng/20 μ L of DNA by QuantiBlot analysis the sample should not be amplified. It can either be re-extracted, reported as containing insufficient DNA or concentrated using a Microcon-100 (see Troubleshooting section). The choice is at the discretion of the interpreting analyst. Other DNA concentrations (especially 0.31 and 0.62 ng/20 μ L) may also be concentrated and purified using a Microcon-100 if the DNA is suspected of being degraded or containing an inhibitor of PCR.
8. After extraction, the tubes containing the unamplified DNA should be transferred to a box and stored in the appropriate refrigerator or freezer. The tubes should *not* be stored in the extraction racks.

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Controls for PCR analysis

The following controls have to be processed along side the sample analysis:

1. A positive control is a DNA sample where the STR alleles for the relevant STR loci are known. The positive control tests the success and the specificity of the amplification, and during the detection and analysis stage the electrophoretical separation and the correct allele calling.
2. An extraction negative control consists of all reagents used in the extraction process, and is necessary to detect DNA contamination of these reagents. A clean result - the absence of detectable DNA - in an extraction negative in one of the PCR multiplexes is sufficient to show the absence of contamination. **Note:** Since the Y STR system only detects male DNA, one cannot infer from a clean Y STR extraction negative the absence of female DNA.
3. An amplification negative control consists of only amplification reagents without the addition of DNA, and is used to detect DNA contamination of the amplification reagents.

Failure of any of the controls does not automatically invalidate the test. Under certain circumstances it is acceptable to retest negative and positive controls. **See STR results interpretation for rules on retesting of control samples.**

Concordant analyses and “duplicate rule”

The general laboratory policy is to confirm DNA results by having concordant DNA results within a case - confirmation of a match or exclusion being the most common situation. Concordant and duplicate analysis is also used to detect sample mix-up and confirm the presence of DNA mixtures.

1. For evidence samples, the following guidelines apply:
 - a. Identical DNA profiles among at least two items (two evidence samples or one evidence sample plus an exemplar) within a case are considered internally concordant results (“duplicate rule”).

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- b. If a sample does not match any other sample in the case, it must be duplicated by amplification in either the same or a second PCR system. If the only result was obtained using Y-STRs, this must be duplicated in the Y system.
- c. If after the first DNA analysis there is an indication that the sample consists of a mixture of DNA, several scenarios need to be considered. Further analysis steps have to be decided based on the nature of each case. Consult your supervisor if you encounter a situation that is not represented in the following examples:
- 1) If all alleles in a mixture are consistent with coming from any of the known or unknown samples in the case, e.g. a victim and a semen source, no further analysis is needed, unless one of the mixture components should be entered in CODIS.
 - 2) If two or more mixtures in a case are consistent with each other and display the same allele combinations, they are considered duplicated.
 - 3) If one or more alleles cannot be accounted for by other contributors in the case, the presence of the foreign component must be confirmed by amplification in either the same or a second PCR system.
 - 4) If there is only one probative profile in a case and this happens to be a mixed sample, the results need to be confirmed by amplification in either the same or a second PCR system depending on the need to type all 13 loci for CODIS.
2. For exemplar samples, the following guidelines apply:
- a. If the DNA profile of a **victim's exemplar** does not match any of the DNA profiles of evidence samples in the case, including mixtures, the victim's exemplar has to be duplicated to eliminate the possibility of an exemplar mix-up. *This is because it is highly likely that an exemplar mix-up would generate a false exclusion.*

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The victim's exemplar must be duplicated starting with a second independent extraction, with the exemplar cut and submitted for extraction by a different analyst. If there is no additional exemplar material available, the duplication may begin at the amplification stage.

Since duplicate exemplar analyses are performed to confirm the exclusion, a partial DNA profile (at least two complete loci) that demonstrates an exclusion is sufficient.

- b. If the DNA profile of a **victim's exemplar** matches any of the DNA profiles of evidence in the case, or is present in a mixture, the exemplar does not have to be duplicated. *This is because it is highly unlikely that a sample mix-up would generate a false inclusion.*
- c. If the DNA profile of a **suspect's exemplar** (or other non-victim elimination exemplar) does not match any of the DNA profiles in the case, or in the local database, the exemplar does not have to be duplicated. *This is meant to streamline the process parallel to convicted offender testing.*
- d. If the DNA profile of a **suspect's exemplar** matches any of the DNA profiles in the case, or in the Local database, the suspect's exemplar has to be duplicated to eliminate the possibility of an exemplar mix-up. *This is meant to streamline the process parallel to convicted offender testing.*

The suspect's exemplar must be duplicated starting with a second independent extraction, with the exemplar cut and submitted for extraction by a different analyst. If there is no additional exemplar material available, the duplication may begin at the amplification stage.

- 3. For evidence samples or exemplar samples analyzed in DNA systems containing overlapping loci, the DNA results for the overlapping loci must be consistent. If no or partial results were obtained for some of the overlapping loci, this amplification is still valid if consistent results were obtained for at least one complete locus. (Amelogenin is not considered an overlapping locus in this context.) If the partial amplification confirms a match or an exclusion of an exemplar or another evidence sample, it does not have to be repeated.
- 4. Partial profiles can satisfy the duplication policy. Consistent DNA typing results from at least one locus in a different amplification (same DNA system or a different DNA system) is considered a concordant analysis.

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5. For Y-STR testing the sample does not have to be reamplified if the internal duplication rule applies or if the Y-STR results are concordant with the autosomal results: confirming an exclusion or inclusion, confirming the presence of male DNA, confirming the number of semen donors. Based on the case scenario it might be necessary to reamplify in order to confirm the exact Y-STR allele calls. There might not be sufficient autosomal data to establish concordance.

DNA storage

1. Store evidence and unamplified DNA in a separate refrigerator or freezer from the amplified DNA.
2. During analysis, all evidence, unamplified DNA, and amplified DNA should be stored refrigerated or frozen. Freezing is generally better for long term storage.
3. Amplified DNA is discarded after the Genotyper analysis is completed.
4. DNA extracts are retained refrigerated for a period of time, then frozen for long-term storage.

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CHELEX DNA EXTRACTION FROM BLOOD AND BUCCAL SWABS

Sample sizes for Chelex extraction should be approximately 3µL of liquid blood or saliva, 1/3 of a swab, or a 3x3mm cutting of a bloodstain.

1. Remove the extraction rack from the refrigerator. Extract either evidence or exemplars. **Do not extract both together.**
2. Pipette 1 mL of sterile deionized water into each of the tubes in the extraction rack.
3. Mix the tubes by inversion or vortexing.
4. Incubate at room temperature for 15 to 30 minutes. This can be done either in a shaker, or by occasionally inverting and vortexing.
5. Spin in a microcentrifuge for 2 to 3 minutes at 10,000 to 15,000 x g.
6. Carefully remove supernatant (all but 30 to 50 µL). If the sample is a bloodstain or swab, leave the substrate in the tube with pellet.
7. Add 175 µL of 5% Chelex (from a well-resuspended chelex solution).
8. Incubate at 56°C for 15 to 30 minutes.
9. Vortex at high speed for 5 to 10 seconds.
10. Incubate at 100°C for 8 minutes using a screw-down rack.
11. Vortex at high speed for 5 to 10 seconds.
12. Spin in a microcentrifuge for 2 to 3 minutes at 10,000 to 15,000 x g.
13. Pipet 20 µL neat and also a 1/10 dilution (using TE⁻⁴) into a microcentrifuge tube for QuantiBlot Analysis to determine human DNA concentration.
14. Store the remainder of the supernatant at 2 to 8°C or frozen.

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CHELEX DNA EXTRACTION FROM SOFT TISSUE (E.G. FETUS SAMPLES)

Sample sizes for this Chelex extraction should be approximately a 3x3mm cutting of tissue.

1. Remove the extraction rack from the refrigerator. Extract either evidence or exemplars. Do not extract both together.
2. Pipette 1 mL of sterile deionized water into each of the tubes in the extraction rack.
3. Mix the tubes by inversion or vortexing.
4. Incubate at room temperature for 15 to 30 minutes. Mix occasionally by inversion or vortexing.
5. Spin in a microcentrifuge for 2 to 3 minutes at 10,000 to 15,000 x g.
6. Carefully remove supernatant (all but 30 to 50 μ L).
7. To each tube add: 200 μ L of 5% Chelex (from a well-resuspended chelex solution).
1 μ L of 20 mg/mL Proteinase K
8. Mix using pipette tip.
9. Incubate at 56°C for 60 minutes.
10. Vortex at high speed for 5 to 10 seconds.
11. Incubate at 100°C for 8 minutes using a screw down rack.
12. Vortex at high speed for 5 to 10 seconds.
13. Spin in a microcentrifuge for 2 to 3 minutes at 10,000 to 15,000 x g.
14. Pipet 20 μ L neat and also a 1/10 dilution and 1/100 (using TE⁻⁴) into a microcentrifuge tube for QuantiBlot Analysis to determine human DNA concentration.
15. Store the remainder of the supernatant at 2 to 8°C or frozen.

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CHELEX DNA EXTRACTION FROM EPITHELIAL CELLS (AMYLASE POSITIVE STAINS OR SWABS, CIGARETTE BUTTS, SCRAPINGS)

Sample sizes for this Chelex extraction should be approximately a 5x5mm cutting or 50% of the scrapings recovered from an item.

1. Remove the extraction rack from the refrigerator.
2. To each tube add: 200 μ L of 5% Chelex (from a well-resuspended chelex solution).
1 μ L of 20 mg/mL Proteinase K
3. Mix using pipette tip.
4. Incubate at 56°C for 60 minutes.
5. Vortex at high speed for 5 to 10 seconds.
6. Incubate at 100°C for 8 minutes using a screw down rack.
7. Vortex at high speed for 5 to 10 seconds.
8. Spin in a microcentrifuge for 2 to 3 minutes at 10,000 to 15,000 x g.
9. Pipet 20 μ L neat and also a 1/10 dilution (using TE⁻⁴) into a microcentrifuge tube for QuantiBlot Analysis to determine human DNA concentration.
10. Store the remainder of the supernatant at 2 to 8°C or frozen.

NOTE: Samples meant for this type of extraction should be placed on the “other evidence” or the Y-STR amylase sheet. Exemplars of this type must be extracted separately.

For very large cuttings 200 μ L of Chelex might not be enough to provide enough suspension of the sample. The reaction can be scaled up and re-concentrated using Microcon concentrators.

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NON-DIFFERENTIAL CHELEX DNA EXTRACTION FROM SEMEN STAINS OR SWABS

Sample sizes for non-differential Chelex extractions depend on the circumstances of the case. Regularly 1/3 of a swab or a 3x3mm cutting of a stain should be used. For cases where semen is present but no sperm cells were detected, the sample size can be increased.

1. Remove the extraction rack from the refrigerator.
2. To each tube add: 200 μ L of 5% Chelex (from a well-resuspended chelex solution).
1 μ L of 20 mg/mL Proteinase K
7 μ L of 1 M DTT
3. Use the pipette tip when adding the DTT to thoroughly mix the contents of the tubes.
4. Incubate at 56°C for approximately 2 hours.
5. Vortex at high speed for 10 to 30 seconds.
6. Incubate in at 100°C for 8 minutes using a screw down rack.
7. Vortex at high speed for 10 to 30 seconds.
8. Spin in a microcentrifuge for 2 to 3 minutes at 10,000 to 15,000 x g.
9. Pipet 20 μ L neat and also a 1/10 dilution (using TE⁻⁴) into a microcentrifuge tube for QuantiBlot Analysis to determine human DNA concentration.
10. Store the remainder of the supernatant at 2 to 8°C or frozen.

NOTE: For very large cuttings 200 μ L of Chelex might not be enough to provide enough suspension of the sample. The reaction can be scaled up and re-concentrated using Microcon concentrators.

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CHELEX DNA EXTRACTION FROM SEMEN STAINS OR SWABS

Approximately 1/3 of a swab or a 3x3mm cutting of a stain should be used for this type of extraction.

1. Remove the extraction rack from the refrigerator.
2. Pipette 1 mL of PBS into each tube in the extraction rack.
3. Mix by inversion or vortexing
4. Incubate at room temperature overnight or for a minimum of 1 hour using a shaking platform.
5. Vortex or sonicate the substrate or swab for at least 2 minutes to agitate the cells off of the substrate or swab.
6. Label new tubes to hold the swab or substrate remains. Remove the swab or other substrate from the sample tube, one tube at a time, using sterile tweezers and close tube. Sterilize tweezers with ethanol before the removal of each sample. Store swab or substrate in a sterile labeled tube for the substrate remains fraction.
7. Spin in a microcentrifuge for 5 minutes at 10,000 to 15,000 x g.
8. Without disturbing the pellet, remove and discard all but 50 µL of the supernatant.
9. Resuspend the pellet in the remaining 50 µL by stirring with a sterile pipette tip.
10. To the approximately 50 µL of resuspended cell debris pellet, add 150 µL sterile deionized water (final volume of 200 µL).
11. Add 1 µL of 20 mg/mL Proteinase K. Vortex briefly to resuspend the pellet.
12. Incubate at 56°C for about 60 minutes to lyse epithelial cells, but for no more than 75 minutes, to minimize sperm lysis.

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13. During the incubation step do the following:
 - a. Label a new tube for each sample, including the extraction negative control. Mark each tube as an epithelial cell fraction
 - b. Add 50 μ L of 20% Chelex (from a well-resuspended chelex solution) to each epithelial cell fraction tube
 - c. Close tubes
14. Spin the extract in a microcentrifuge at 10,000 to 15,000 x g for 5 minutes.
15. Add 150 μ L of the supernatant from each sample and the extraction negative to its respective epithelial cell fraction sample tube. Store at 4°C or on ice until step 21.
16. Wash the sperm pellet with Digest Buffer as follows:
 - a. Resuspend the pellet in 0.5 mL Digest Buffer.
 - b. Vortex briefly to resuspend pellet.
 - c. Spin in a microcentrifuge at 10,000 to 15,000 x g for 5 minutes.
 - d. Remove all but 50 μ L of the supernatant and discard the supernatant.
 - e. Repeat steps a-d for a total of 5 times.
17. Wash the sperm pellet once with sterile dH₂O as follows:
 - a. Resuspend the pellet in 1 mL sterile dH₂O.
 - b. Vortex briefly to resuspend pellet.
 - c. Spin in a microcentrifuge at 10,000 to 15,000 x g for 5 minutes.
 - d. Remove all but 50 μ L of the supernatant and discard the supernatant.
18. Resuspend the pellet by stirring with a sterile pipette tip.
19. To the approximately 50 μ L resuspended sperm fraction and to the tubes containing the substrate remains and the sperm fraction extraction negative, add 150 μ L of 5% Chelex, 1 μ L of 20 mg/mL Proteinase K, and 7 μ L of 1M DTT. Mix gently.
20. Vortex both the epithelial cell and sperm fractions. The following steps apply to all fractions.
21. Incubate at 56°C for approximately 60 minutes.

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22. Vortex at high speed for 5 to 10 seconds.
23. Incubate in at 100°C for 8 minutes using a screw down rack.
24. Vortex at high speed for 5 to 10 seconds.
25. Spin in a microcentrifuge for 2 to 3 minutes at 10,000 to 15,000 x g.
26. Pipet 20 µL neat and also a 1/10 dilution (using TE⁻⁴) into a microcentrifuge tube for QuantiBlot Analysis to determine human DNA concentration.
27. Store the remainder of the supernatant at 2 to 8°C or frozen.

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CHELEX DNA EXTRACTION FROM HAIR

Microscopic examination of hair by PD lab should be completed before attempts at DNA typing. If possible document state of root on photo. Be careful not to loose adhering sheath material prior to extraction.

A. Sample preparation for loose hairs:

1. Handling hair with clean forceps, examine the hair under a dissecting microscope for the presence of sheath material. The hair may be placed on a clean piece of white paper. Note possible presence of body fluid on hair.
2. Wash the hair containing sheath material to remove surface dirt and contaminants by immersing the hair in sterile, deionized water in a clean 15 or 50 mL Falcon tube.
4. Return the hair to the dissecting microscope. Use a clean scalpel to cut a 1 cm portion from the root end of the hair and place in a 1.5 mL microcentrifuge tube.

B. Sample preparation for mounted hairs:

5. Remove cover slip by placing slide in freezer for 10 minutes and afterwards removing cover with a razor blade or clean forceps. Alternatively the slide can be submerged in fresh xylene for 1-2 minutes.
6. Add 10-20 μ L of fresh Xylene to the embedded hair sample. Allow 10-20 seconds for Permount to dissolve, then tease out the hair sample with clean forceps. Check slide for tissue that was left behind.
7. Place hair on clean microscope slide and add more fresh Xylene to remove adhering Permount. Pipet off Xylene, add a few drops of water, repeat rinsing with water several times.
8. Put hair under dissecting microscope. Use a clean scalpel to cut a 1 cm portion from the root end of the hair and place in a 1.5 mL microcentrifuge tube.

ATTENTION: If not immediately starting Chelex extraction, do not place wet hair in 1.5mL tube. Perform additional 99% Ethanol rinse and let hair dry before placing in tube.

ATTENTION: Do not consume hair shaft. If possible leave at least 1cm for mtDNA typing.

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C. Chelex extraction

9. To hair tubes add: 100 μ L of 5% Chelex (from a well-resuspended chelex solution).
1 μ L of 20mg/mL Proteinase K.

Note: Check that the hair is completely immersed in the Chelex solution before incubating.

10. Incubate at 56°C for at least 6 to 8 hours, or overnight.
11. Vortex at high speed for 5 to 10 seconds.
12. Incubate at 100°C for 8 minutes using a screw down rack.

Note: Check that the hair is completely immersed in the Chelex solution before boiling.

13. Vortex at high speed for 5 to 10 seconds.
14. Spin in a microcentrifuge for 2 to 3 minutes at 10,000 to 15,000 x g.
15. Pipet 20 μ L neat and also a 1/10 dilution (using TE⁻⁴) into a microcentrifuge tube for QuantiBlot Analysis to determine human DNA concentration.
16. Store the remainder of the extract at either 2 to 8°C or frozen.

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ORGANIC EXTRACTION PROCEDURE

A. Sample Preparation

Liquid/dry blood, bone marrow, oral swab and tissue sample preparation

Stained substrates and oral swabs should be cut into small pieces (3 x 3 mm). Tissues should be minced into small pieces in a weigh boat using a new razor blade. Place samples in 1.5mL microcentrifuge tubes or conical tubes when appropriate. See table below for various sample types.

Proceed to Section B: Sample Incubation

Sample type	Amount
Liquid blood	100 to 500 μ L
Bone marrow	0.5 x 0.5 cm to 1.5 x 1.5cm
Oral swab	1/3 to a whole swab
Blood stain	0.5 x 0.5 cm to 1.5 x 1.5cm
Soft tissue	0.5 x 0.5 cm to 1.5 x 1.5cm
Paraffin embedded tissue	0.3 x 0.3 cm to 1.0 x 1.0 cm

Bone preparation

Before extraction, a bone or tooth specimen should be cleaned entirely of soft tissue and dirt using a range of methods, such as scraping, rinsing and sonication. A combination of sterile scalpels, sterile toothbrushes and running water should be used to clean. For a sonication bath, the sample is placed in a conical tube and covered with a 5% Terg-a-zyme solution. For additional cleaning, the sonication step may be repeated multiple times by decanting the liquid and replacing with fresh Terg-a-zyme solution. After cleaning, the sample is usually rinsed with distilled water and dried using a 56°C incubator (drying time may vary from a few hours to overnight).

Note: Terg-a-zyme is an enzyme-active powdered detergent. A 5% solution should be made fresh prior to bone preparation and cleaning. Refer to pg. A78, Appendix A in the QA Manual. Once prepared, the reagent will only be effective for up to 16 hours.

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1. Photograph bone or tooth sample after cleaning. Measure and weigh specimen prior to sampling.
2. If several bones are available, generally compact bone is preferred, such as humerus, femur, or tibia.

****WARNING****

Protective eyewear, lab coats, cut resistant gloves and HEPA-filtered facial masks should be worn when cutting bone. Avoid breathing bone dust. All cutting of bone must be done under a biological hood.

3. Using an autopsy saw or a Dremel tool equipped with a 409 or 420 cutting wheel, cut the bone specimen into approximately 5x5x5mm size pieces. Take enough cuttings for an end weight of approximately 2g. For older or compromised bones, several aliquots of 2g can be extracted and combined during the Microcon step. For tooth samples, the whole root should be taken. **Note: The cut off wheel should be disposed of after each use.**
4. Place bone cuttings in 50mL conical tubes labeled with the FB case number, ME#, PM item #, initials, and date.
5. Cover bone cuttings with 5% Terg-a-zyne solution and sonicate samples for 30-45 minutes. **Note: Ensure water level in the sonicator is 1-2 inches from the top.**
6. Decant the Terg-a-zyne and wash with distilled water until no detergent bubbles remain.
7. If necessary, repeat with fresh changes of 5% Terg-a-zyne and water washes until the dirt has been removed.
8. Place the clean cuttings in a weigh boat on a small Kim Wipe. Cover with another weigh boat. Label the weight boat with the FB case number, ME#, PM item #, initials, and date.
9. Seal with evidence tape.
10. Dry in a 56°C incubator for a few hours or overnight. After sufficient drying, weigh bone cuttings. **The bone sample must be completely dry before milling.**

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Sample milling with the SPEX Certiprep 6750 Freezer Mill

All freezer mill parts that come into contact with bone specimens, such as the cylinders, metal end plugs and impactors, should be cleaned, dried and sterilized prior to use. See Step 20 for appropriate cleaning procedure.

1. Assemble specimen vials in the following order: metal bottom, plastic cylinder, impactor, and metal top.
2. Place under UV light for a minimum of 15 minutes.
3. Label metal bottoms with blue Sharpie ink with a case identifier.
4. Add bone cuttings to specimen vial around impactor using decontaminated forceps. Cover with metal top. **Note: Shake specimen vial and ensure that the impactor can move back and forth.**
5. Wipe down inside of mill with a wet paper towel. **Do not use bleach or ethanol.**
6. Plug in mill and switch ON.
7. Obtain liquid nitrogen from tank by filling transfer container. Be aware that the liquid nitrogen tank may be empty when the detector level reads anywhere from “¼” to “empty”.

****WARNING****

Liquid Nitrogen can be hazardous. Use cryogenic gloves, protective eyewear/face shield and lab coats when handling. Avoid liquid nitrogen splashes to face and hands.

8. Open the freezer mill lid. Add liquid nitrogen slowly into the mill up to the **FILL LINE** to avoid splashing and boiling over.
9. Place the specimen vial into the round chamber. If processing more than one bone sample it is possible to save pre-cooling time by placing up to two vials in the mesh container inside the mill.
10. Change cycle number to match total number of samples plus two ($n + 2$).

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11. Adjust mill settings as follows:

Cycle	set to # of samples + 2
Time	T1 (milling) 2.0 min T2 (pause) 2.0 min T3 (pre-cool) 15.0 min
Rate	Bones – 8-10 Teeth – 6-8

12. Close cover slowly to avoid any liquid nitrogen splashes and press **RUN** to start the mill. Pre-cooling will begin followed by the milling cycle.
13. During the 2-minute pause phase, it is now possible to open the mill and remove the finished sample using cryogenic gloves.
14. Place one of the pre-cooled specimens waiting in the dock in the round chamber.
15. If liquid nitrogen level is below the **FILL LINE**, refill. A loud noise during milling means that the liquid nitrogen level is low. If liquid nitrogen is not refilled, damage to the mill, mill parts, and cylinder can occur.
16. Close the lid and press **RUN** again. Repeat from Step 11 until all samples are processed.
17. Inspect each sample after removal from the mill. If sample is sufficiently pulverized, remove the metal top using the Spex Certi-Prep opening device.
Note: Samples may be reinserted into the mill for additional grinding.
18. Using decontaminated tweezers, remove impactor from vial and submerge in 10% Bleach.
19. Empty bone dust into labeled 50mL Falcon tube. Ensure complete dust transfer by tapping bottom of cylinder. Weigh bone dust and document.
20. Soak metal end parts and plastic cylinder in 10% Bleach.
21. When milling is complete, switch mill to **OFF** and unplug. Leave cover open for liquid nitrogen to evaporate. The next day, lower cover and place in storage until next use.

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22. **Mill Parts -Clean Up:** Mill parts must be cleaned immediately after processing. If this is not possible, steps a-b must be completed before leaving overnight.
- Rinse off 10% bleach.
 - Soak all parts in 0.1% SDS.
 - Brush parts with a new toothbrush to remove any residual bone dust.
 - Rinse with water.
 - Soak parts in 10% bleach and brush each part in bleach individually.
 - Rinse with water.
 - Separate the plastic cylinders from the metal parts.
 - Rinse in 100% ethanol. **ONLY** the metal top, metal bottom, and compactor can be rinsed in 100% ethanol. **DO NOT** rinse the plastic cylinder in ethanol as it will cause the plastic cylinder to break.
 - Use isopropanol to remove any identifying marks made with a Sharpie on the tops or bottoms of the cylinders.
 - Dry and expose the parts to UV light for a minimum of 2 hours. The UV light in a biological hood or a StrataLinker can be used.
23. Proceed to Section B: Sample Incubation.

Laser Microdissection

This procedure can be used to separate maternal (decidual) tissue from fetal material (chorionic villi) in early stage abortions. The procedure can only be performed in cooperation with the OCME histology department and a trained pathologist. The instrument used, the PixCell® Iie Laser Capture Microdissection System (ARCTURUS Systems for Microgenomics, Carlsbad, CA), is owned by NYU Medical center and can only be operated by trained NYU staff.

1. Initial processing

The product of conception (POC) can be received in different stages of preparation:

- POC scrapings in saline buffer:

Remove tissue from liquid either by filtration or centrifugation:

- Transfer liquid to 50mL falcon tube
- Spin sample in a bench top Eppendorf or IEC Centra CL3R at 1000 RPM for 5 minutes
- Discard liquid supernatant

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Submit sample to the Histology department for tissue processing according to the OCME Histology Procedure Manual section E. Then proceed as for b.).

b) POC fixated and embedded in paraffin blocks:

Contact histology department and ask them to prepare microscope slides from the paraffin block using the following precautions:

- Use disposable blades for the microtome and discard after each case.
- Clean working surface on microtome by wiping with 10% bleach and alcohol before and after each case.
- Use individual floating chambers for each case
- Use uncharged microscope slides

The slides then should be stained with hematoxylin and eosin-phloxine (H&E technique) as described in the OCME Histology Procedure Manual. But again during the staining procedure separate sets of jars have to be used for each case.

c) Stained or unstained microscope slides from POC blocks:

If the slides are unstained, ask the histology department to stain them as described above. Otherwise proceed with the microdissection technique. **Attention:** for slides that were prepared by a histology laboratory outside of the OCME, foreign DNA not from the mother and the fetus might be present on the slide.

2. PixCell IIe Laser Capture Microdissection

The PixCell® IIe Laser Capture Microdissection System (LCM) utilizes a low-power infrared laser to melt a special thermoplastic film over the area of interest. Especially developed CapSure Caps that are coated with this thermoplastic film are placed on the tissue sample and are used to capture the cells of interest after the laser excitation. See the Arcturus instrument manual for a more detailed description.

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A trained pathologist has to be present to distinguish decidual tissue from chorionic villi and operate the laser. After the slide has been placed on the microscope platform the pathologist will visually identify the area of interest, mark this area for the laser, and activate the laser. The laser setting is specified in the Arcturus instrument manual. The Forensic Biology Criminalist needs to be present during the complete procedure to maintain chain of custody of the evidence.

An area of chorionic villi and an area of maternal tissue should be collected on separate CapSure caps. The caps can be stored and transported in 50 ml Falcon tubes. A third unused CapSure cap should be extracted as an extraction negative control.

Use new scalpel and clean forceps to remove the film from the cap and transfer the film to a fresh 1.5mL reaction tube containing 500µL of organic extraction buffer, DTT, SDS and Proteinase K as described below.

B. Sample Incubation

1. Process an extraction negative with every batch of extractions.
2. Prepare the master mix and mix thoroughly by swirling or vortexing *very briefly*.

For liquid blood, dry blood and bone marrow samples:

	1 Sample	5 Samples	10 Samples	15 Samples
Organic extraction buffer	400 µL	2.0 mL	4.0 mL	6.0 mL
20% SDS	10µL	50 µL	100µL	150 µL
Proteinase K (20 mg/mL)	13.6 µL	68 µL	136 µL	204 µL
Total Incubation Volume <i>per sample</i> :				400 µL

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For bone samples:

	Per bone (~2g dust)	1 sample (N+ 2)	3 samples (N+ 2)	5 samples (N+ 2)
Organic Extraction Buffer	2370 µL	7.11 mL	11.85 mL	16.59 mL
20% SDS	300 µL	900 µL	1.5 mL	2.1 mL
0.39 M DTT	120 µL	360 µL	600 µL	840 µL
Proteinase K (20 mg/mL)	210 µL	630 µL	1.05 mL	1.47 mL
Total Incubation Volume <i>per sample</i> :				3000 µL

For teeth samples:

	Per tooth	1 sample (N+ 2)	3 samples (N+ 2)	5 samples (N+ 2)
Organic Extraction Buffer	790 µL	2.37 mL	3.95 mL	5.53 mL
20% SDS	100 µL	300 µL	500 µL	700 µL
0.39 M DTT	40 µL	120 µL	200 µL	280 µL
Proteinase K (20 mg/mL)	70 µL	210 µL	350 µL	490 µL
Total Incubation Volume <i>per sample</i> :				1000 µL

For tissues and paraffin embedded tissue (e.g. microdissection) samples:

	Per tissue	1 sample (N+ 2)	3 samples (N+ 2)
Organic extraction buffer	395 µL	1185 µL	1975 µL
20% SDS	50 µL	150 µL	250 µL
0.39 M DTT	20 µL	60 µL	100 µL
Proteinase K (20 mg/mL)	35 µL	105 µL	175 µL
Total Incubation Volume <i>per sample</i> :			500 µL

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3. Add appropriate incubation volume of master mix to each tube. Vortex briefly. Make certain the substrate, tissue, or swab is totally submerged. **Note: Reagent volumes may be adjusted in order to accommodate the size or nature of a particular sample.**
4. Place tubes in a shaking 56°C heat block and incubate overnight.
5. Proceed to Section C: Phenol Chloroform Extraction and Microcon[®] cleanup.

For semen stains and swabs

1. Make sure that you process an extraction negative control for both cell fractions.
2. Stains up to 1.5 x 1.5 cm can be extracted. Trim excess fabric from stain. If a stain is very diffuse or is on a bulky substrate, the extraction can be scaled up appropriately. For swabs remove cotton swab from applicator stick with a razor blade.
3. Cut stained fabric or swab into small pieces (3x3 mm).
4. Place fabric or swab into a 1.5 mL microcentrifuge tube.
5. Prepare a master mix for N+2 samples as follows:

	1 Sample	5 Samples	10 Samples
Digest buffer	400 µL	2.0 mL	4.0 mL
Proteinase K (20 mg/mL)	5 µL	25 µL	50 µL

6. Add 400 µL of master mix to each sample.
7. Place the tubes in a 37°C heat block and incubate for 2 hours.
8. Make sure the caps are on tight. Using a clean, sterile needle, punch a hole in the bottom of each tube (heating the needle makes punching the hole much easier).

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9. Piggyback the tube containing the sample inside a new labeled tube. Collect the lysate by centrifuging at 2700 x G in a swinging bucket rotor for 2 minutes. Save the tube containing the fabric or swab until after DNA quantification results were obtained. Optional- If the substrate remains are to be re-extracted for additional bound sperm, transfer the fabric or swab to a new marked tube.
10. Spin the extract in a microcentrifuge at 10,000 to 15,000 x g for 5 minutes.
11. Without disturbing the pellet, pipet 350 µL of the supernatant in into a new labeled tube. This supernatant is called the epithelial cell fraction and is enriched for non-sperm DNA. Place the epithelial cell fraction on ice until step 17. In this step, the negative control is treated like a sample even though it should not have a pellet. The sperm negative is derived from the remaining lysate in the negative control and the epithelial negative is derived from the 350µL of supernatant, which was pipetted off.
12. Wash the sperm pellet with Digest buffer as follows:
 - a. Resuspend the pellet in 0.5 mL Digest Buffer.
 - b. Vortex briefly to resuspend pellet.
 - c. Spin in a microcentrifuge at 10,000 to 15,000 x g for 5 minutes.
 - d. Remove all but 50 µL of the supernatant and discard the supernatant.
 - e. Repeat steps a-d for a total of 3 times.
13. Resuspend the pellet in the remaining liquid.
14. Prepare a master mix for N+2 samples as follows:

	1 Sample	5 Samples	10 Samples
Digest Buffer	400 µL	2.0 mL	4.0 mL
0.39 M DTT	40 µL	200 µL	400 µL
Proteinase K (20 mg/mL)	10 µL	50 µL	100 µL

15. Add 450 µL of the master mix to each pellet and optionally to the substrate remains.
16. Place the tubes in a 37°C heat block and incubate for 2 hours.

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17. Proceed to Section C: Phenol Chloroform Extraction and Microcon[®] cleanup.

C. Phenol Chloroform Extraction and Microcon Clean up

Set Up

Remove the Phenol:Chloroform:Isoamyl Alcohol (25:24:1) (PCIA) from the refrigerator. Aliquot from the lower, clear organic layer. Alternatively, if the PCIA is allowed to equilibrate to room temperature, swirl thoroughly to form a single homogenous phase. Unscrew cap and aliquot desired amount.

Obtain organic waste jug for disposal of any tubes or pipette tips that come in contact with PCIA.

See Section D for preparation of Phase Lock Gel (PLG) tubes.

For samples possibly needing mtDNA or Low Copy Number DNA testing: Place one Microcon[®] YM100 collection tube and one 1.5 mL microcentrifuge tube for each sample, including the extraction negative, in the StrataLinker for at least 15 minutes. **Note: Irradiate multiple tubes (4-6) per bone sample to accommodate the total volume of incubation buffer.**

Extraction

1. Vortex and centrifuge incubated microcentrifuge tube samples at high speed for 1 minute. Vortex and centrifuge bone dust, incubated in 50 mL conical tubes, for 5-10 minutes at 1000 RPM in Eppendorf Centrifuge Model 5810.
2. Obtain one Eppendorf Phase Lock Gel (PLG) tube per sample, including the extraction negative. PLG tubes make phase separation easier and are optional. **Note:** For bone samples, label as many tubes to accommodate the total volume of incubation buffer per sample. For example, if you incubated 2g of bone dust with 3 mL of incubation buffer, you will technically need 6 PLG tubes.
3. Centrifuge PLG tubes at maximum speed for 30 seconds.
4. Label Microcon[®] YM100 filters for each sample. Prepare the Microcon[®] YM100 concentrators by adding 100 μ L of TE⁻⁴ to the filter side (top) of each concentrator. Set aside until step 11.

****WARNING****

Phenol Chloroform is toxic. Protective eyewear, mask, lab coat, and nitrile gloves should be worn when handling. All work must be conducted under a chemical fume hood.

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5. Add a volume of Phenol:Chloroform:Isoamyl Alcohol 25:24:1 (PCIA) equal to the volume of incubation buffer (typically 400 µL) to be added from the sample to the PLG tube. **Note: When pipetting PCIA, you must penetrate the top oil layer. Place used pipette tips in the organic waste bottle.**
6. Have someone witness your samples, PLG, and Microcon[®] YM100 tube set up.
7. Pipette the sample supernatant (typically 400 µL) to the PLG tube already containing PCIA. For bone dust samples, pipette several aliquots of the supernatant into multiple PLG tubes. **Note: Do not disturb bone pellet.**
8. Shake the PLG tube vigorously by hand or by inversion to form a milky colored emulsion. **Note: Do NOT vortex the PLG tube.**
9. Centrifuge samples for 2 minutes at maximum speed to achieve phase separation. (On Eppendorf Centrifuge Model 5415D, spin at 16.1 RCF or 13.2 RPM).
10. If the sample is discolored, contains particles in the aqueous phase, or contains a lot of fatty tissue, transfer the top layer (aqueous phase) and repeat Steps 7-9. Note: The aqueous layer from bone and teeth will usually be discolored. Only repeat extraction steps if any dust or particles are present in the aqueous layer. If it is not necessary to repeat the extraction step, go to Step 11.
11. Carefully transfer the aqueous phase (top layer) to the prepared Microcon[®] YM100 concentrator. Be careful not to let the pipette tip touch the gel. **Note: Discard used PLG tubes into the organic waste bottle.**
12. Spin the Microcon[®] YM100 concentrators for 15 minutes at 500 x g, which is approximately 2500 RPM. (On Eppendorf Centrifuge Model 5415D, spin at 0.6 RCF or 2600 RPM). **Note: Ensure that all fluid has passed through filter, if not, spin for additional time, in 5-minute increments. If fluid still remains, collect sample and microcon again using new filter.**
13. Discard the wash tubes and place the concentrators into a new collection tube.
14. Add 400 µL of TE⁻⁴ to the filter side of each Microcon[®] YM100 concentrator.
15. Spin again at 500 x g, which is approximately 2500 RPM, for 15 minutes.

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16. Add 40 μL of TE^{-4} to the filter side of each Microcon[®] YM100 concentrator. **Note: For bone samples, add only 10-20 μL of TE^{-4} to each filter side to ensure smallest elution volume.**
17. Invert sample reservoir and place into a new labeled collection tube. (*For samples possibly needing mtDNA or Low Copy Number DNA testing, invert sample reservoirs into irradiated collection tubes*). Spin at 1000 x g, which is approximately 3500 RPM, for 3 minutes to collect sample. (On Eppendorf Centrifuge Model 5415D, spin at 1.2 RCF or 3600 RPM).
18. Measure the approximate volume recovered and record on the organic extraction worksheet. **Note: Combine bone elutants before measuring volume.**
19. Discard sample reservoir and adjust sample volume depending on the starting amount and expected DNA content as follows using TE^{-4} . **Note: Samples may be microcon'ed again to further concentrate low DNA content samples.**

Sample type	Final Volume
High DNA content (Large amounts of blood, fresh tissue, bone marrow, oral swabs, and dried bloodstains)	400 μL
Medium DNA content (Small amounts of blood, fresh tissue, bone marrow, oral swabs, and dried bloodstains); differential lysis samples	200 μL
Low DNA content (Formalin fixed tissue, dried bone, teeth, samples from decomposed or degraded remains, some reference samples)	100 μL

20. Transfer samples to newly labeled 1.5mL microcentrifuge tubes for storage. (*For samples possibly needing mtDNA or Low Copy Number DNA testing, transfer samples to irradiated 1.5 mL microcentrifuge tubes*). Record the approximate final volume on the organic extraction worksheet.
21. Submit aliquots for QuantiBlot or Real Time PCR analysis to determine human DNA concentration. Follow the procedures for submitting extracts to either quantitation method.

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22. Store at 2 to 8°C or frozen.

NOTE: For larger volumes, 15 ml PLG tubes may be prepared as well as Centricon[®] YM100 concentrators (available in 2mL, 15mL and other sizes). For concentration, spin at 500x g and at 1000x g for recovery. Adjust RPM according to centrifuge used. Do not exceed 1000x g. Please be aware that using the larger PLG and the Centricon[®] YM100 concentrators will require longer spin times.

NOTE: See Microcon[®] troubleshooting.

D. Preparation of Phase Lock Gel (PLG) tubes

The purchase of Phase Lock gel (PLG) Syringes Heavy is economical and more convenient than having to stock all possible tube sizes. The gel in the syringe can be added to different disposable tubes. Make sure the plasticware being used is resistant to phenol and chloroform.

1. Without putting pressure on the plunger, twist off the **orange cap** and discard. Attach the **gray dispensing tip** (supplied) to the syringe and tighten securely. (NOTE: Use of gray tip is optional for a smoother application of PLG. Less force is necessary when gray tip is NOT used.)
2. Apply firm pressure on the plunger to dispense PLG until it reaches the end of gray tip.
Add heavy PLG based on Table below.
NOTE: 325µL = 3.25 cc corresponds to 3 lines on the syringe

Tube size	PLG heavy
0.5mL	100µL
1.5mL	325µL
2.0mL	325µL
15mL	3mL
50mL	5mL

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3. Pellet the PLG by spinning the tubes prior to use. See table below.

Tube size	Centrifuge model	Speed	Time
0.5 to 2.0mL	Eppendorf 5415C Eppendorf 5415D	14 x 1000 RPM 13.2 x1000 RPM/16.1 x1000 RCF	30 seconds
15 and 50mL	Sigma 4-15 C	1500 RCF	2 minutes

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MICROCON 100 DNA CONCENTRATION AND PURIFICATION

Microcon 100 filter membranes have a pore size that retains all DNA fragments >100bp. This filtration device can be used to concentrate a low DNA sample (e.g. when the quantification reads <0.15ng/20µL) by simply removing excess liquid, or to purify a sample from low molecular weight contaminants (e.g. if the QuantiBlot is inconclusive or an amplification inhibitor is suspected). After the liquid reduction the volume might be as small as 2-5µL, it is therefore necessary to bring the volume back up by adding TE⁻⁴. In order to allow duplicate amplifications the final volume shouldn't be smaller than 50µL.

1. Label a sufficient number of blue Microcon 100 sample reservoirs and insert them into the vials.
2. Fill out Microcon worksheet. Process 50µL of TE⁻⁴ solution as an Microcon negative control. Make sure to use the same lot that is used to dilute the samples and don't forget to label the final negative control tube with the Microcon date and time and your initials.
3. Pipette 100 µL of TE⁻⁴ solution into each labeled sample reservoir including the negative control. Add DNA sample (0.4 mL maximum volume) to buffer. Don't transfer any Chelex beads, or in case of an organic extraction sample any organic solvent! Seal with attached cap. ***Avoid touching the membrane with the pipette tip!***

Return the original Chelex tubes to their storage location. Do not discard empty tubes.

4. Place the assembly into a variable speed microcentrifuge. Make sure all tubes are balanced! ***To prevent failure of device, do not exceed recommended g-forces.***

Spin at 500 x g (2500 RPM, Eppendorf) for 15 minutes at room temperature.

5. For **purification** of the DNA sample add 200 µL of TE⁻⁴ solution and repeat Step 4. Do this as often as necessary to generate a clear extract, and then continue with Step 6. When performing multiple wash steps it is necessary to empty the bottom vial intermittently.

For **concentration** only proceed to step 6.

NOTE: When purifying samples with a low DNA concentration it might be advantageous to use several wash steps and to also reduce the volume to achieve both, a cleaner sample and an increased DNA concentration.

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6. Remove assembly from centrifuge. Open attached cap and add 20 μ L TE⁻⁴. ***Avoid touching the membrane with the pipette tip!*** Separate vial from sample reservoir.
7. Place sample reservoir upside down in a new **labeled** vial, then spin 3 minutes at 1000 x g (3500 RPM Eppendorf) for 3 minutes. Make sure all tubes are balanced!
8. Remove from centrifuge and separate sample reservoir. Measure resulting volume using an adjustable Micropipette, record volume on work sheet; adjust volume to desired level using TE⁻⁴.

Clean-up for high DNA concentrations: reconstitute starting volume.
Low DNA samples (clean-up and concentration): adjust to 50 μ L.

Transfer the DNA extracts and the microcon negative control to new labeled 1.5mL Eppendorf tubes and store extract for later use. Note storage location on worksheet.
9. Calculate resulting concentration or submit 10 μ L for QuantiBlot.

ATTENTION: Do not store the DNA in the Microcon vials! The lids are not tight enough to prevent evaporation.

Troubleshooting:

Lint, bone dust and other particles can clog the membrane. If the liquid does not go down, redistribute supernatant to multiple vials. Pipet off clear supernatant without disturbing particle pellet.

If the problem persists, the specific Microcon lot number might be faulty. Notify the QA Unit and try a different lot number.

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MAGATTRACT DNA EXTRACTION FROM BLOODSTAINS AND BUCCAL SWABS

Sample size for the extraction should be approximately 1/3 of a swab or a 3x3 mm cutting of the stain. **This extraction is applicable to bloodstain and buccal swabs only, NOT cigarette butts.**

All bloodstain and exemplar cuttings should be placed in 2.0mL screw cap sample tubes. All samples in 1.5mL Eppendorf tubes should be transferred to a 2.0mL screw cap sample tube prior to extraction.

Add samples to the appropriate M48 Sample Submission Sheet- exemplars or evidence. Include sample info, tube label, target date and IA initials. The analyst performing the extraction will assign sample rack position numbers.

A. Setting up M48 Spreadsheet and Saving Sample Name List

1. Collect the M48 Sample Submission Sheets for the extraction. On these sheets, assign each sample a sample rack position, remembering that the extraction negative will occupy Position 1. Also fill in the initials of the analyst performing the extraction and the extraction date and time. This date and time will be used throughout the extraction.
2. Open the appropriate M48 spreadsheet, evidence (M48EV) or exemplar (M48EX) depending on your sample set.
2. Click the "Input Sample Names" tab and enter the sample names for the extraction, including the extraction negative, into the appropriate positions in column B.
3. Save this sheet by going to File → Save As and save the sheet to the "SampleName" folder on the desktop with "File Name:" in MMDDYY.HHMM format and "Save As Type:" set to CSV (Comma delimited)(* .csv). For instance an extraction performed at 2:20pm on May 23, 2006 would be saved, with date and time in military format, as 052306.1420.csv. This ensures proper data basing of sample sheets and report pages.
4. Click "Save".
5. A window stating "The selected file type does not support workbooks that contain multiple sheets" will open. Click "OK".

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6. A second window asking “Do you want to keep the workbook in this format?” opens. Click “Yes”.
7. Click the Ext Sheet 24 or Ext Sheet 48 tab depending on the batch size of the extraction. For batches consisting of 24 or less samples, including the extraction negative, use Ext Sheet 24. For batches consisting of 25-48 samples, including the extraction negative, use Ext Sheet 48.
8. Once the appropriate extraction sheet is open, finish the sheet by entering the tube label, target date, and IA initials for each sample.
9. Print the extraction sheet by going to File → Print. Verify the selected printer is Lexmark Z52 Series Colorfine and click “OK”. Leave the Control Program window for the printer open. It will disappear after printing is complete.
10. **Minimize** the M48 spreadsheet.

B. Sample Preparation and Incubation

1. Remove the extraction rack from the refrigerator. Extract either evidence or exemplars. Do not extract both together.
2. Wear appropriate lab gear for all handling of samples and reagents particularly when working over the robotic platform. This includes gloves, lab coat, and mask. Sample preparation should be performed under a hood.
3. Have a witness verify your samples.
4. For large runs, prepare master mix for N+2 samples as follows, vortex briefly, and add 200uL to each of the tubes in the extraction rack and the pre-prepared extraction negative tube. For smaller runs, you may add Proteinase K and G2 Buffer to each tube individually:

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Reagent	1 sample	6 samples	12 samples	18 samples	24 samples
Digestion Buffer (Buffer G2)	190 µL	1520 µL	2660 µL	3800 µL	4940 µL
QIAGEN Proteinase K	10 µL	80 µL	140 µL	200 µL	260 µL

- Shake at 1000 rpm at 56° C for a minimum of 30 minutes.

C. BioRobot M48 Software and Platform Set-Up

- Double click on the “BioRobot M48” icon on the desktop.
- Click the “Start” button. **Note: The door and container interlock must be closed to proceed.**
- “F Trace MTL” protocol should be selected. If not, click on the arrow in the middle of the screen and then select “New Dev” → “gDNA” → and “F Trace MTL”.
- Click on the “select” button and select “1.5 ml” for the size of the elution tubes.
- Select the number of samples 6, 12, 18, 24, 30, 36, 42, or 48.
- Set sample volume to 200 uL (can not and should not change).
- Set elution volume to 200 uL.
- The next prompt asks to ensure the drop catcher is clean. In order to check this click on “manual operation” and select “Drop Catcher Cleaning”. The arm of the robot will move to the front of the machine, and the drop catcher (a small plastic tray) will be right in front of you. Remove and clean with **ethanol**. When the catcher is clean, replace the tray, close the door, and click “OK” in the window.
- Place a bag for the tips to be discarded. Click “Next”.

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10. The software will calculate the number of tips necessary for the run. Place tips in the tip rack(s) if necessary. **When filling racks make sure that the pipette tips are correctly seated in the rack and flush with the robotic platform.** Tips are located in three racks. These racks may be filled one at a time, BUT you must fill a **whole rack** at a time. After a rack is filled, reset the tip rack by clicking on “Yes tip rack ...”. If no new tips are being added to the robot click “No”.

NOTE: When opening a new tip bag, ALL tips should be placed onto the robotic platform. Open tip bags should not be returned to the drawer. Racks may be used for tip storage. When adding tips, spilling into the next empty rack is OK, just do not **reset** the rack until it is **completely** full.

Tips needed for a run:

# Samples	6	12	18	24	30	36	42	48
# Tips	30	42	54	66	78	90	102	114

After you are finished, click “Next”

11. Fill the reagent reservoirs as stated below. All reagents are stored in their respective plastic reservoirs in the metal rack, covered with Parafilm, **EXCEPT** the magnetic resin. The resin is stored between runs in its original stock bottle to prevent evaporation. Vortex the magnetic resin solution well, both in the stock bottle and in the reservoir, before adding it to the metal rack. If you notice crystallization in any of the solutions, discard the solution, rinse the container out, and start again with fresh reagent.
12. Remove the parafilm and lids from the reagents, and fill the reservoirs to the appropriate level using solutions from the working solution bottles, **adding approximately 10% to the volumes recommended below to account for the use of the large bore pipette tips:**

Note: Do not forget that bottles of MW1 require the addition of ethanol prior to use. See bottle for confirmation of ethanol addition and instructions for preparation if needed.

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# of samples	Large reservoir Sterilized Water (mL)	Large reservoir Ethanol (mL)	Large reservoir Buffer MW1 (mL)	Large reservoir Buffer MTL (mL)	Small reservoir Buffer MW2 (mL)	Elution buffer (TE ⁻⁴) (mL)	Small reservoir Magnetic Resin (mL)
6	10.0	11.8	7.2	5.9	3.5	2.5	1.5
12	18.4	22.6	12.9	10.3	5.9	3.7	1.7
18	26.9	33.4	18.6	14.7	8.4	4.9	1.9
24	35.3	44.2	24.3	19.0	10.8	6.1	2.1
30	43.7	55.0	30.0	23.4	13.3	7.3	2.3
36	52.2	65.8	35.7	27.8	15.7	8.5	2.5
42	60.6	76.6	41.4	32.1	18.2	9.7	2.7
48	69.0	87.4	47.0	36.5	20.6	10.9	2.9

Place into the metal rack in the following locations. The plastic reservoirs only fit into the rack one way. Check the directions of the notches which should point **into** the robot:

Size Container	Rack Position	Software Tag	Reagent
Large Container	L4	Rea_4	Sterilized Water
Large Container	L3	Rea_3	Ethanol (100%)
Large Container	L2	Rea_2	Wash Buffer 1 (Buffer MW1)
Large Container	L1	Rea_1	Lysis and Binding Buffer (Buffer MTL)
Small Container	S6	ReaS6	(empty)
Small Container	S5	ReaS5	(empty)
Small Container	S4	ReaS4	(empty)
Small Container	S3	ReaS3	Wash Buffer 2 (Buffer MW2)

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Size Container	Rack Position	Software Tag	Reagent
Small Container	S2	ReaS2	Elution Buffer (TE ⁻⁴)
Small Container	S1	ReaS1	Magnetic Particle Resin

13. Flip up the “container interlocks” and place the metal reservoir holder onto the left side of the robotic platform in the proper position. **DO NOT force the holder into place and be careful not to hit the robotic arm.** After correctly seating the metal holder, flip down the “container interlocks” and press “next”.
14. Click “Next” when you are prompted to write a memo.
15. Place the sample preparation trays on the robot. One tray for every 6 samples. Click “Next”.
16. Place empty, unlabeled 1.5mL elution tubes in the 65 degree (back) hot block, located on the right side of the robotic platform. Click “Next”.
17. Label 1.5 mL screw top tubes for final sample collection in the robot.
18. Place **labeled**, empty 1.5 mL sample collection tubes in the 8 degree (front) cold block for collection of final samples.
19. At this point, the samples should be near the end of the incubation period (From Section A, Step 5). Spin all tubes in a microcentrifuge for 1 minute at 10,000 to 15,000 x g. **When they are ready, have a witness confirm the order and labels of both the sample tubes and the labeled 1.5 mL final sample collection tubes. The robot setup witness should also verify that all plastic-ware is in the correct position and correctly seated in the platform.**
20. Remove caps and place the samples for extraction on the robot. Discard the caps. **For empty positions, add a 2.0 mL sample tube filled with 200 uL of sterile water.**
21. Click “Yes” when asked to input sample names.

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D. Importing Sample Names

1. At the sample input page, click “Import”.
2. The Open window will appear. “Look in:” should automatically be set to a default of “SampleName”. If not, the correct pathway to the folder is My Computer\C:\Program Files\GenoM-48\Export\SampleName. (The SampleName folder on the desktop is a shortcut to this file.
3. Select your sample name file and click “Open”. Verify that your sample names have imported correctly. Do not be concerned if a long sample name is not completely displayed in the small window available for each sample.
4. Manually type in the word “Blank” for all empty white fields.
5. Click “Next”.

E. Verifying Robot Set-Up and Starting the Purification

1. In addition to confirming the *position* of all plasticware and samples, check the following conditions before proceeding:

All plasticware (tips, sample plates, tubes) is seated properly in the robotic platform	✓
Metal reservoir rack is seated properly, UNDER the interlocks	✓
Interlocks are down	✓
Sample tubes, elution tubes and sample collection tubes have been added to the platform in multiples of 6 as follows:	
Empty 1.5 mL tubes are filling empty positions for both sets of elution tubes in the cold and hot blocks	✓
2.0 mL sample tubes filled with 200uL of sterile H2O are in empty positions of the sample rack	✓

2. After confirming the position and set-up of the plastic-ware click “Confirm”.
3. Click “OK” after closing the door.

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- Click “Go” to start the extraction.
- The screen will display the start time, remaining time, and the completion time. You are now free to complete other work until the extraction is complete.
- It is advisable to monitor the extraction until the transfer of DNA sample from the sample tubes to the first row of sample plate wells to ensure proper mixing of magnetic resin and DNA sample.
- At the end of the extraction, a results page will be displayed indicating the pass/fail status of each set of six samples. See Section F for instructions for printing out the report page.

F. Saving and Printing Extraction Report Page

- At the results page click the “Export” button at the bottom center of the screen. The Save As window will appear. “Save In:” should be set to the “Report” folder on the desktop. This is a shortcut to the following larger pathway: My Computer\C:\Program Files\GenoM-48\Export\Report.
- In “File Name:”, name the report in the format MMDDYY.HHMM. Set “Save As Type:” to Report Files (*.csv). For instance an extraction performed at 4:30pm on 5/14/06 would be saved as 051406.1630.csv.
- Click “Save”.
- Maximize the M48 spreadsheet by clicking its icon on the bottom tool bar.
- At the bottom of the spreadsheet, click the “Import Run Results” tab.
- Highlight cell “A1” and in the pull-down menus go to Data → Get External Data → Import Text File...
- In the Import Text File window select:

Look in: Report (For specific pathway refer to Section F Step 1)
Files of Type: All files
File Name: Select your extraction run results by date and time
- Click “Import”.

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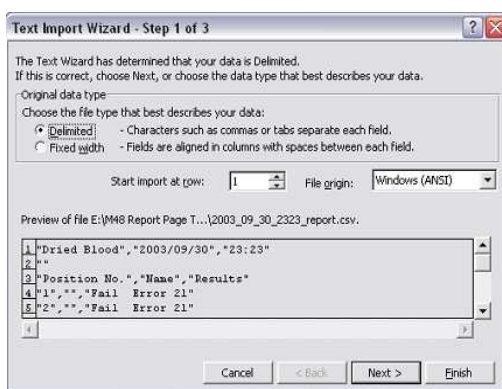
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9. In the Text import Window Step 1 of 3, check the following settings:

Original Data Type: Delimited
Start Import at Row: 1
File Origin: WINDOWS (ANSI)

The window should appear as below:



10. Click "Next".

11. In Text Import Window Step 2 of 3, select the following:

Delimiters: Place a check by comma. Make sure no other options are checked.
Text qualifier: "

Verify that the settings and data preview corresponds to those in the window below:



12. Click "Next".

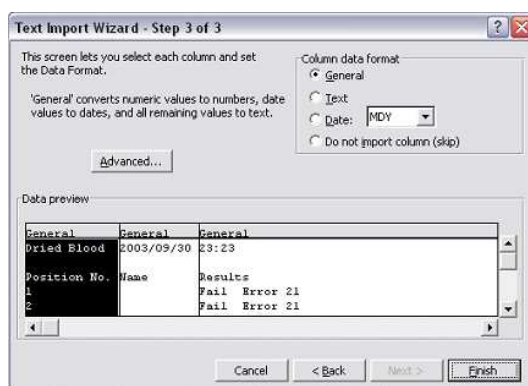
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13. In Text Import Window Step 3 of 3, select the following:

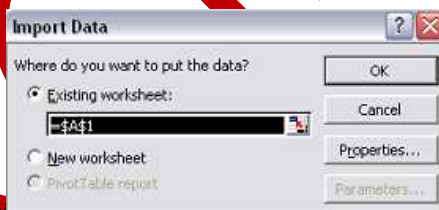
Column Data Format: General

The window should appear as below:



14. Click “Finish”.

15. In the Import Data window “Existing Worksheet” should be selected and the data input cell should read “=A\$1”. See below:



16. Click “OK”. Data w

17. Click on the “Report” tab and verify that the run data has correctly imported into the report page.

18. Manually enter the analyst’s initials and extraction date (MM/DD/YY) and time (HH:MM AM/PM) in the highlighted cells.

19. Print the run report page.

20. Close the spreadsheet by going to File → Exit. A window asking “Do you want to save changes you made to...?”. Click “No”.

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21. Proceed with clean-up and sterilization.

G. Post-Extraction Clean Up and UV Sterilization

1. Wipe down the robotic platform and waste chute with **Ethanol. DO NOT USE SPRAY BOTTLES.**
2. Discard used pipette tips, sample tubes, and sample preparation plate(s).
3. Replace the lid on the magnetic resin reservoir and vortex remaining resin thoroughly. Transfer the Magnetic resin to the stock bottle immediately with a 1000µL pipetteman. Rinse the reagent container with de-ionized water followed by ethanol and store to dry.
4. Cover all other reagents and seal with Parafilm for storage. **DON'T FORGET TO LABEL RESERVOIRS WITH THE LOT NUMBER OF THE REAGENT THEY CONTAIN.**
5. Click "Next".
6. When prompted, "Do you want to perform a UV sterilization of the worktable?", click "Yes".
7. Select 1 Hour for the time of "UV sterilization" then click "yes" to close the software upon completion.
8. Have a supervisor sign off on the run report and submit samples for quantitation, neat, 1/10, and/or 1/100 dilutions, as needed.
9. **DON'T FORGET TO FILL OUT THE M48 USAGE LOG WITH THE TIME AND DATE OF THE EXTRACTION, USER INITIALS, AND ANY COMMENTS ARISING FROM THE RUN.**

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H. BioRobot M48 Platform Diagram

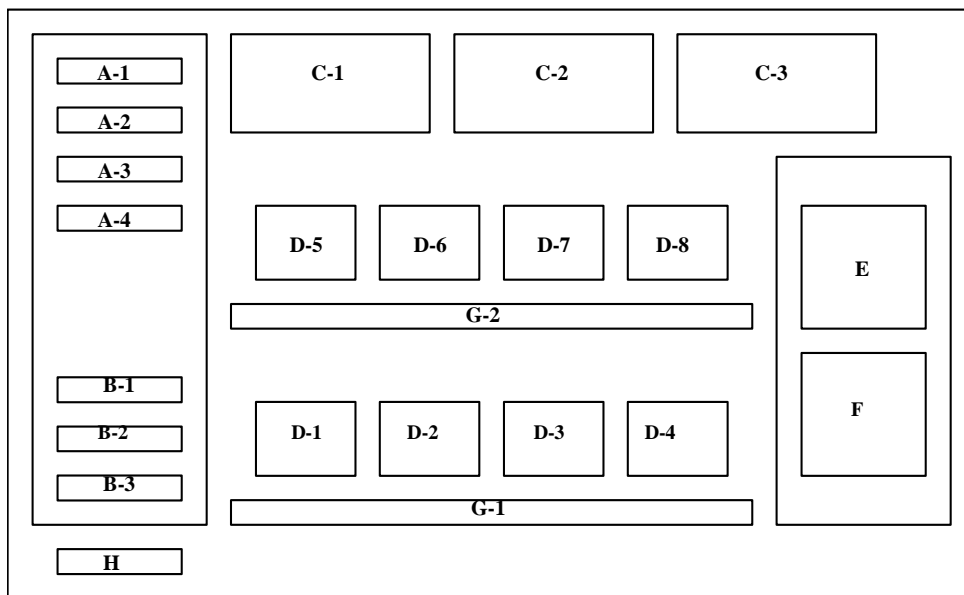


Figure 1. Diagram of Robotic Platform of the QIAGEN BioRobot M48.

- A (1-4) Large Reagent Reservoir Positions
- B (1-3) Small Reagent Reservoir Positions
- C (1-3) Tube Racks 1, 2, and 3
- D (1-8) Sample Plate Holders
- E Hot Elution Block (65 degrees)
- F Cold Final Elution Block (8 degrees)
- G (1-2) Sample Tube Racks
- H Waste Disposal Chute

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I. Troubleshooting

ERROR	CAUSE/REMEDY
Resin/sample is being drawn up into pipette tips unequally	Report problem to QA. Resin buffer has evaporated. O-rings are leaking and need service.
Crystallization around 1 st row of wells in sample plate	Forgot to fill empty sample tubes with 200uL of sterile H ₂ O.
BioRobot M48 cannot be switched on	BioRobot M48 is not receiving power. Check that the power cord is connected to the workstation and to the wall.
Computer cannot be switched on	Computer is not receiving power. Check that the power cord is connected to the computer and to the wall power outlet.
BioRobot M48 shows no movement when a protocol is started	BioRobot M48 is not switched on. Check that the BioRobot M48 is switched on.
BioRobot M48 shows abnormal movement when a protocol is started	The pipettor head may have lost its home position. In the QIAsoft M software, select “ <u>M</u> anual Operation/ Home”.
Aspirated liquid drips from disposable tips.	Dripping is acceptable when ethanol is being handled. For other liquids: air is leaking from the syringe pump. Report problem to QA. O-rings require replacement or greasing. If the problem persists, contact QIAGEN Technical Services

Revision History:

May 26, 2006 – 1) Revised “Organic Extraction Procedure” for clarification. 2) Added “MagAttract DNA Extraction From Bloodstains and Buccal Swabs.” See [Approval Form](#).

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Sample Blotting

1. Vortex DNA Standards and Calibrators 1 and 2. Centrifuge briefly to bring the contents to the bottom of the tube. If Chelex extracts are being used, centrifuge for 2 minutes.
2. While wearing gloves, label enough microfuge tubes for all standards. Labels can be made on the tops of the microfuge tubes, and should be made to reflect the wells that they will be loaded in (i.e. 10 ng standard - 1A, 5 ng standard - 1B, etc.).

Pipet standards into the microfuge tubes, using the following amounts of each:

- a. DNA Standards and Calibrators - 5 μ L
 - b. Negative (TE^{-4}) - 5 μ L
3. While wearing gloves, label the tops of the sample tubes. Labels should be made in the same manner as for the standards. The samples were aliquoted ahead of time by the chelex rotation and are stored at 4°C in the QuantiBlot refrigerator.
 4. Label a hybridization tray with the date, rack # of the samples, and your initials. Should the weekly allotment of trays be depleted, it is permissible to wash used trays containing membranes that have **already been reviewed** by the rotation supervisor. These trays should be thoroughly washed with ethanol and rinsed with deionized water. Labels may be removed with isopropanol. The trays should then be dried completely with a kimwipe prior to use.
 5. Heat a shaking water bath to 50°C. The water level should be 1/4 to 1/2 inch above the shaking platform. The temperature should not go below 49°C or above 51°C. **It is essential to check the temperature with a calibrated thermistor probe or thermometer before the hybridization is performed. Also remember to record the temperature.**

Heat a stationary water bath to between 37°C and 50°C. Warm the QuantiBlot Hybridization Solution and the QuantiBlot Wash Solution in the water bath. **All solids must be in solution before use.**

6. Once you begin the rest of the QuantiBlot assay, you must finish. Allow approximately 3 hours.

Add 150 μ L of Spotting Solution to each tube. Vortex and centrifuge briefly to bring the contents to the bottom of the tube.

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7. Obtain a QuantiBlot membrane from the drawer and place it in a hybridization tray containing 50 mL of Pre-Wetting Solution and incubate at room temperature for 1-30 minutes. Note: Either side of the Biodyne B membrane can be used as the side onto which samples are pipetted. **Always wear gloves when handling the membrane.**
8. Inspect the gasket wells to make sure they are aligned correctly (improper alignment can lead to “thin” bands upon color development). Then, using forceps, remove the membrane from the Pre-Wetting solution. Place the membrane on the gasket of the slot blotter, then place the top plate of the slot blotter on top of the membrane. Turn on vacuum pump to a vacuum pressure of approximately 200 to 250 mm Hg. Turn off the sample vacuum and turn on the clamp vacuum on the slot blot apparatus. Push down to ensure a tight seal.
9. At this point have someone witness the tube setup.
10. Load the membrane as follows:

Using a new pipet tip for each sample, apply all of each sample into a separate well of the slot blotter. For best results, slowly dispense each sample directly into the center of the wells, with the pipet tip approximately 5 mm above the membrane. **Note: Do not allow the pipet tip to touch the membrane since this may compromise the membrane at that spot.**

SLOT	SAMPLE
1A	10 ng Standard
1B	5 ng Standard
1C	2.5 ng Standard
1D	1.25 ng Standard
1E	0.625 ng Standard
1F	0.3125 ng Standard
1G	0.15625 ng Standard
1H	Plate negative Control (TE ⁴)
2A	3.5 ng Calibration 1 Standard
2B	0.5 ng Calibration 1 Standard
2C	0.15625 ng Standard
2D – 6F	Samples and Controls
6G	3.5 ng Calibration 1 Standard
6H	0.5 ng Calibration 1 Standard

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11. After all the samples have been applied, **slowly** turn on the sample vacuum. Leave the sample vacuum on until all samples have been drawn through the membrane. Inspect each slot that contains a sample for a uniform blue band. If a uniform blue band is not visible, make a note of it.

Turn off the sample vacuum, the clamp vacuum, then the vacuum source.

12. Disassemble the slot blotter and remove the membrane. Proceed immediately to pre-hybridization. Do not allow the membrane to dry out.

Clean the apparatus by soaking in enough 0.1% SDS to cover for 5-15 minutes. Following soaking in SDS, rinse the gasket and the side of the top plate that contacts the membrane **thoroughly** with H₂O (include a final rinse with deionized H₂O). Make sure that all the wells of the top plate are rinsed and clean. Rinse the bottom unit of the slot blotter with deionized H₂O and allow to dry at room temperature. **Never use bleach.** A toothbrush can be used to clean the surfaces if necessary.

13. Transfer the membrane to 100 mL of pre-warmed QuantiBlot Hybridization Solution in the hybridization tray. Add 5 mL of 30% H₂O₂. Place the lid on the tray. Put the tray into the 50°C shaking water bath. Place a weight (e.g. lead ring) on the covered tray to prevent the tray from sliding or floating.

Shake at 50°C for 15 minutes at 50-60 rpm. Pour off the solution.

Hybridization

14. Add 30 mL of pre-warmed QuantiBlot Hybridization Solution to the tray. Tilt the tray to one side and add 20 µL of QuantiBlot D17Z1 Probe to the QuantiBlot Hybridization Solution. Cover tray with lid and weight.

Shake at 50°C for 20 minutes at 50-60 rpm. Pour off the solution.

15. Add 100 mL of pre-warmed QuantiBlot Wash Solution to the tray. Rinse by rocking for several seconds, then pour off the solution.

16. Add 30 mL of pre-warmed QuantiBlot Wash Solution to the tray. Tilt the tray to one side and add 180 µL of Enzyme Conjugate. Cover tray with lid and weight.

Shake at 50°C for 10 minutes at 50-60 rpm. Pour off the solution.

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17. Add 100 mL of QuantiBlot Wash Solution to the tray. Rinse by rocking for 1 minute, then pour off the solution. Repeat for a total two washes.
18. Add 100 mL of QuantiBlot Wash Solution to the tray. Cover tray with lid and weight.

Shake at room temperature for 15 minutes at 100-125 rpm. Pour off the solution. During this time, prepare the Color Development Solution (see below).

Color Development

19. In a glass flask, prepare the Color Development Solution. Add the reagents in order:

60 mL of Citrate Buffer
3 mL Chromogen
60 µL 3% H₂O₂.

Mix thoroughly by swirling (do not vortex).

Note: Do not prepare the Color Development Solution more than 10 minutes before use. Use a new tube of hydrogen peroxide for each batch of Color Development Solution. Discard the remaining hydrogen peroxide after use. Wrap the Chromogen bottle in Parafilm after each use to prevent oxidation.

20. Add 100 mL of Citrate Buffer to the tray. Rinse by rocking for several seconds, then pour off the solution.
21. Add the Color Development Solution to the tray. Cover tray with lid.

Develop the membrane by shaking at room temperature for 20-60 minutes at 50-60 rpm. Pour off the solution.
22. Stop the color development by washing in approximately 100 mL deionized H₂O. Repeat several times. After the last wash, store membrane in deionized H₂O. Cover tray with lid and proceed with photography.

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Photography

23. Photograph the membrane while wet. Place the membrane on a dark, flat, non-absorbent surface.
24. Use a Polaroid MP4 camera system with type 667 or 664 film and a Wratten 23A or 22 (orange) filter.
25. Turn on the flood lights. Adjust the height of the camera and focus so that the membrane fills the entire viewing frame.
26. Photograph at 1/125 seconds and f8 for type 667 film. Photograph at 1/8 seconds and f8 for type 664 film.
27. Develop the film for 30-60 seconds. If the photograph is out of focus, not exposed properly, or does not accurately record the bands on the membrane, vary the exposure conditions and re-photograph.

Indicate the appropriate columns and rows on front of the photograph. Also, initial and date the photograph.

28. Attach the photograph to the QuantiBlot worksheet. Once the QuantiBlot passes review (see below), make photocopies and distribute to the appropriate analysts. File the original QuantiBlot worksheet along with photograph in the appropriate binder. Discard the membrane.

Note: If an entire QuantiBlot or a portion of it does not pass review (see below), it is the responsibility of the analyst on the QuantiBlot rotation to resubmit those samples that are in question.

Quality Control

1. The DNA standards should yield band intensities that decrease approximately two-fold for each subsequent dilution (see below for exceptions).

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2. Compare the intensities of calibrator 1 (3.5 ng) and calibrator 2 (0.5 ng) to those of the DNA standards. The intensity of calibrator 1 should be between 2.5 and 5 ng; the intensity of calibrator 2 should be between 0.31 and 0.62 ng. Calibrators 1 and 2 are loaded in duplicate on the membrane. At least three of these four calibrator samples must be consistent with the DNA ladder and each other. **If more than one of the calibrator samples are incorrect, the membrane must be repeated.**
3. Estimate the quantity (ng) loaded for each sample by comparing band intensities of the unknown samples with those of the DNA standards. If the band intensity is between two DNA standards, **match the sample to the DNA standard with the lower amount of DNA.**

It is recommended to run the neat and 1/10 dilution for each sample. As a general rule in determining the DNA quantity of an unknown sample, the band intensity must be less than the largest DNA standard (10 ng). If the sample band intensity is ≥ 10 ng, multiply the reading of the 1/10 dilution by 10 to determine the DNA quantity for that sample. If the sample band intensity of the 1/10 dilution is ≥ 10 ng, then submit 1/100 and 1/1000 (if necessary) dilutions for analysis. To determine the DNA quantity for these samples, multiply the 1/100 and 1/1000 dilution readings by 100 and 1000, respectively.

4. **Membranes that have compromised DNA standards may pass review, however, care must be taken in the interpretation of the results.** Examples of compromised DNA standards include bands that are not visible, nonuniform signal intensity within a slot, and samples that yield a band intensity that is not consistent with that of the other DNA standards.

The following guidelines must be followed:

- a. At least one of the 0.15 ng standards must be visible. If not, samples that show band intensities of < 0.31 ng and samples with no apparent DNA must be repeated, including the extraction negative control(s).
- b. If one of the DNA standards other than 0.15 ng produces a band intensity that is not consistent with the other DNA standards and the DNA calibrators, then sample readings that fall at or near the amount of the compromised standard, and are between the adjacent two standards (one on either side of the compromised standard) are not valid for that membrane.

For example, if the 2.5 ng DNA standard is compromised, the sample readings that are < 5 ng but > 1.25 ng are not valid for that membrane. If the 10 ng DNA standard is compromised, sample readings > 5 ng are not valid for that membrane.

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- c. If more than one of the DNA standards are compromised, the membrane can pass review if four consecutive DNA standards are correct as determined by comparing band intensities to those of the DNA calibrators. In such a case, the membrane will be valid for the determination of DNA sample amounts that yield values at or within those four DNA standards.

NOTE: this scenario only applies if the 10ng and the 0.15ng standard fails. All other standards are required to read the calibrators.

5. If the membrane fails review for the determination of DNA amounts, it can still be used as a guide in the resubmission of neat samples and/or dilutions. Also, any bands that are produced on the membrane are indicative of the presence of human DNA*, provided that the corresponding plate negative and extraction negative controls do not produce a signal.
6. Submit a photograph and QuantiBlot Worksheet for review by the QuantiBlot Station supervisor.

* Non-human primate DNA may give comparable results to that of human DNA using this procedure (Perkin Elmer Corp. 1996).

Quantiblot interpretation

1. The Quantiblot procedure, done properly, gives reproducible and fairly accurate determinations of the total amount of human DNA. The neat sample and 1/10 dilution should give quantitation results that make sense - the neat and 1/10 samples should correlate with one another.

Neat	1/10 Dilution	1/10 Dilution
	Results OK	Review photo to determine best concentration
≥10	5, 2.5, 1.25	0.62
5	0.62	1.25, 0.31
2.5	0.31	0.62, 0.15
1.25	0.15	0.31, Blank
0.62	<0.15	0.15
0.31	Blank	<0.15
0.15	Blank	N/A

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2. If you have any pairs of results other than those above, you need to take steps to correct the problem **before** amplification. A review of the photograph may be all that is needed to determine the discrepancy of the results (e.g. incorrectly called results, non-uniform signal intensity, thick or thin bands) and obtain a reasonable estimate of the DNA concentration.
 1. If neat and 1/10 dilution are both ≥ 10 ng, submit additional dilutions for Quantiblot.
 2. If neat and 1/10 dilution are of equal intensity, resubmit neat and 1/10 dilution for Quantiblot.
 3. If neat and 1/10 dilution are too far apart in intensity (e.g. 5 and 0.15 ng, 2.5 ng and blank), resubmit neat and 1/10 dilution for Quantiblot.
 4. If neat and 1/10 dilutions are both “*” due to colored impurities, then the sample may need cleaning up using a Microcon spin filter followed by Quantiblot.

Modified QuantiBlot starting with extracts in microtiter plates

For this procedure the number of controls and calibrators has been reduced so that multichannel pipettes can be used for sample loading. All steps are performed as above with the following modifications:

1. Add 150 μ L of Spotting Solution to the prepared standards and to each sample well. Do not vortex microtiter plate! Proceed without delay.
2. Prepare membrane and gasket as described above.
3. Load the membrane as follows:

Slot	Sample
1A	10 ng standard
1B	5 ng standard
1C	2.5 ng standard
1D	0.625 ng standard
1E	0.3125 ng standard
1F	0.15625 ng standard
1G	Plate negative control (TE ⁻⁴)
1H	3.5 ng calibration standard

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4. Load samples in columns 2A-H to 6A-H using an 8-channel 300 µL pipette. Proceed as described above.
5. Due to the reduced number of standards and calibrators the acceptance guidelines were changed as follows:
 - a.) The intensity of the 3.5ng calibrator has to be between 2.5 and 5ng, otherwise the membrane must be repeated.
 - b.) The membrane passes review if at least three standards in a row are correct and their intensities decrease in linear fashion.

Troubleshooting of QuantiBlot

(Taken from the Applied BioSystems QuantiBlot package insert, see QuantiBlot references)

<u>Observation</u>	<u>Possible Cause</u>	<u>Recommended Action</u>
1. No signal or low sensitivity.	Use of a membrane other than Biodyne B.	Use Biodyne B nylon membrane. Do not use membranes that have neutral charge.
	Incorrect NaOH or EDTA concentrations in Spotting Solution.	Prepare Spotting Solution correctly.
	Water bath temperature too high.	Water bath temperature should be 50°C (± 1°C).
	DNA Probe was not added at hybridization step.	Add QuantiBlot D17Z1 Probe.
	Enzyme conjugate was not added.	Add Enzyme conjugate: HRP-SA at indicated step in protocol. Use 180 µL of Enzyme Conjugate: HRP-SA.
	Hydrogen peroxide was inactive.	Prepare a new Color Development Solution using a fresh bottle of hydrogen peroxide.

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<u>Observation</u>	<u>Possible Cause</u>	<u>Recommended Action</u>
	Presence of $MgCl_2$ in the DNA sample.	Concentrations of $MgCl_2$ >0.3 mM can result in reduced sensitivity. Prepare all DNA dilutions in TE^{-4} Buffer. Any $MgCl_2$ can be removed from samples by micro dialysis using Microcon 100 spin units (follow manufacture's directions).
2. Areas of low sensitivity across the membrane.	Membrane slipped up onto the side of the Hybridization Tray during Hybridization or Stringent Wash steps.	Reduce the rotation rate of the water bath to 50-60 rpm. Check that the membrane is fully submerged in the bottom of the Hybridization Tray before shaking.
	Membrane dried-out significantly at some point in the protocol.	Do not allow the membrane to dry at any point in the protocol.
3. Non-uniform signal intensity within a slot.	Bubbles(s) in slot blot wells when sample was pipetted into well, or when vacuum was applied.	Slowly pipet the Spotting Solution directly over the center of the wells of the slot blot apparatus, with the pipet tip raised approximately 5 mm above the membrane.
		Turn on the sample vacuum slowly, not all at once. After being drawn through the membrane, the sample should appear as a uniform blue band on the membrane.

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<u>Observation</u>	<u>Possible Cause</u>	<u>Recommended Action</u>
		If the entire sample is not drawn through the membrane, turn off the sample vacuum. Pipet the sample back into the pipet tip; then pipet the sample back into the well of the slot blot apparatus. Turn on the sample vacuum to draw the sample through the membrane.
4. Filter background	No or low SDS in the Hybridization Solution or in the Wash Solution	Prepare solutions with proper concentrations of SDS.
	Membrane was not pre-wetted prior to slot blotting.	Pre-wet the membrane in Pre-Wetting Solution prior to slot blotting.
	Too much Enzyme conjugate: HRP-SA was added.	Use 180 μ L of Enzyme Conjugate:HRP-SA.
	Lack of thorough rinsing.	Thoroughly rinse twice, for 1 minute each, using 100 mL of pre-warmed Wash Solution. These two rinse times can be extended beyond 1 minute if necessary.
	Slot blot apparatus not cleaned thoroughly after last use.	Immediately after each use, soak the slot blot apparatus in a large volume of 0.1% SDS solution. Never use bleach.
5. The DNA Calibrators do not quantitate correctly with respect to the DNA Standards.	DNA Standard serial dilutions prepared incorrectly.	Prepare two-fold serial dilutions of DNA Standard in TE ⁻⁴ Buffer as described. Add 5 μ L of each dilution to 150 μ L of Spotting Solution for slot blotting.

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<u>Observation</u>	<u>Possible Cause</u>	<u>Recommended Action</u>
6. Signal obtained for non-human DNA samples.*	Water bath temperature too low.	Water bath temperature should be 50°C (± 1°C).
	SSPE concentration too high in Wash Solution.	Check that the 20X SSPE solution and the Wash Solution were prepared correctly.

- * DNA from primate species may give signals similar to those obtained from equivalent amounts of human DNA. In Roche Molecular Systems (RMS) laboratories, 30 ng to 300 ng quantities of non-primate DNA samples result in either no signals or signals that are less than or equal to the signal obtained for 0.15 ng of human DNA.

Revision History:

October 5, 2006 – Section re-numbered to “3A” to insert Section 3B (Real Time PCR Methods). See [Approval Form](#).

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ALU-Based Quantitative Real Time PCR on the Rotorgene

A. Paperwork Preparation

The aim of the following steps is to set up an Excel spreadsheet with a list of samples that can be used for quantitation and facilitate PCR set up. LCN (low copy number) samples are contact DNA samples with low expected DNA yields, while HCN (high copy number) samples are body fluid samples.

1. Open the “RG summary sheet” excel file template in the Rotorgene - RG sheets folder.
2. In cell D3 type in the name of the quantitation assay as follows: RG# and “Q” or the name of the RG, month, day, and year, period, hour and minutes. For example, RG1Q040905.1330 or Gerty051707.1500.
3. Exemplar and evidentiary samples may be quantitated simultaneously. However, exemplar extracts must be diluted prior to performing the assay. In other words, only the aliquots and/or dilutions of the exemplars may be present with the evidentiary samples under the hood.
NOTE: Since LCN evidence samples are diluted only when necessary and since LCN extract volume is limited such that only the two required microliters can be spared for quantitation, extreme care must be used in pipetting the LCN quantitation aliquot in its entirety.
4. Determine which samples require quantitation. Prioritize samples accordingly.
5. Copy/Type sample names from the extraction sheet and/or “to be quanted sheet”, and paste into the Rotorgene “sample sheet” (second sheet of the excel workbook). For samples requiring dilutions, the dilution factor should be entered in decimal form following a comma after the sample name. For instance, for bloodstain 1A a 1/10 dilution is required. This sample should be entered into the RG sample sheet as “bloodstain 1A, 0.1”.

NOTE: To facilitate import of sample information into the 3130xl software, if sample info is copied or imported at the level of the extraction sheet, the sample info should already adhere to 3130xl software requirements. INCLUDE REQUIREMENTS. This would similarly apply to the copying or importation of sample info at the level of the RG sheet. Letters, numbers, and only the following characters: - _ . () { } [] + ^ may be used. Do not use commas (except to separate sample and dilution info), colons, or quotes. Use the character ^ instead of quotes.

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6. Three calibrators and 15 standards are measured with each assay; therefore, 54 samples may be measured on each RG assay.
7. If applicable, enter the initials of the analyst's to whom paperwork should be directed, the target date, and the top tube label under the "IA", "target date", and "tube label" columns, respectively. If not available or not applicable, type a dash in the cell. For quant results going directly to the analyst rather than the auto-alliquot system, enter an "A" as a superscript to the analyst's initials.
8. On sheet one, the "assay sheet", in cell D4, enter the name of the extraction assay. If multiple extraction sets are being run, enter "misc".
9. The number of samples that are being measured will be automatically calculated and shown in cell E7 as samples are added to the "sample sheet". Verify that this number is correct. The spreadsheet will automatically calculate how much of each reagent to aliquot.
10. Save the sheet in the appropriate Rotorgene folder using the quantitation name. (Ex. Quantitation assay RG1Q040905.1300 will be saved in the RG1 folder as "RG1Q040905.1300".)
11. Print the assay sheet, as well as the sample sheet by clicking File and Print on the menu bar. This step must be repeated for both sheets independently.

B. Personal Preparation

1. Follow gowning protocol procedures.
For example, in the LCN laboratory this includes the following:
 - a. Put on a mask, lab coat, hair covering and gloves.
 - a. Ensure that the cuffs of the lab coat completely cover arms.
 - b. Lab coats may be recycled.
2. Do not enter room without appropriate attire. Moreover, never touch any apparatus, bench etc with bare hands.
3. Remove gloves when answering the phone and touching door handles in order to maintain a DNA free glove.

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C. Work Place Preparation

1. Apply 10% bleach followed by water, and 70% ETOH to the entire work surface, cap opener, and pipets. **All Rotorgene setup steps should be carried out under a hood.**
2. Retrieve clean racks, cap openers, Rotorgene 0.1 mL tubes and caps, microcentrifuge tubes, and irradiated GIBCO™ ULTRA PURE™ distilled water from storage or the stratalinker.
 - a. The tube racks and the cap opener may be cleaned with 10% bleach followed by ETOH. Ensure that the racks are dry before use.
 - b. The 1.5 mL microcentrifuge tubes and water aliquots in 1.5 mL tubes must be irradiated for 30 and 45 minutes, respectively.
 - c. The Rotorgene tubes and caps are used as packaged.
3. Arrange work place to minimize crossover. For example, the sharps waste for tips should be on one side whereas the fresh filter pipet tips and reagents should be on the opposing side.
4. Position gloves nearby with bleach/ETOH/water in order to facilitate frequent glove changes and cleaning.

D. Sample Dilution

If necessary, for HCN DNA samples for example, dilute the sample extracts.

1. Label microcentrifuge dilution tubes with sample name and dilution.
2. Place each dilution tube directly behind the corresponding extract tube in a rack.
3. Add the appropriate amount of diluant (irradiated water or TE) to each dilution according to Table 1.
 - a. Sexual assault semen and saliva samples, scrapings and other samples that are extracted with the “Chelex other” method, and bone samples should be measured with a neat and a 1/100 dilution.
 - b. Blood and buccal samples and all burglary samples may be measured with a 1/10 dilution only. This will capture most concentrations. If necessary, a second measurement may be taken with either a neat or a 1/100 dilution.
 - c. LCN samples should be measured with a neat dilution. If necessary, a 1/10 dilution may be made if one suspects inhibition.
 - d. Pipet tips do not need to be changed to add water/TE to empty tubes. Close all caps.

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- Open only one sample and its corresponding dilution tube at one time.
- Thoroughly mix each extract, prior to aliquotting.
- Immediately following each dilution, return the original sample extract tube to its cyrobox. Return the original samples to 4°C storage.
- Once the dilutions are completed, evidentiary samples may join exemplar dilutions under the hood.

TABLE 1:

	Submission 1			Submission 2		
	Dilution 1	Sample	Water or TE	Dilution 2	Sample	Water or TE
HCN Semen and saliva (amylase positive) samples	Neat	5 µL	0	1/100	2 µL	198 µL
HCN Scrapings or "other" extractions	Neat	5 µL	0	1/100	2 µL	198 µL
HCN exemplars Bone	Neat	5 µL	0	1/100	2 µL	198 µL
HCN exemplars Blood or Saliva	1/10	2 µL	18 µL	1/100 or neat (if necessary)	2 µL or N/A	198 µL or N/A
HCN Blood Samples	1/10	2 µL	18 µL	1/100 or Neat (if necessary)	2 µL or n/A	198 µL or N/A
Touched objects and/or LCN Samples	Neat	N/A	N/A	1/10 (if necessary)	2 µL	18 µL

For LCN DNA samples, the neat LCN sample may be taken from the extract tube and added to the quantitation tube directly. (No neat submission tube is necessary.) However, 1/10 dilutions should be prepared in advance as specified above.

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E. Remove reagents for the master mix from the reagent freezer/refrigerator

1. Retrieve MgCl_2 , 10X PCR buffer, BSA, dNTPs, TAQ GOLD, unlabeled “EB1” and “EB2” primers, and SYBR Green I from the freezer, irradiated GIBCO™ ULTRA PURE™ distilled water from the refrigerator, and DMSO from the cabinet.
2. Store reagents except DMSO and water in a Nalgene cooler on the bench.
3. Record lot numbers of reagents.
4. Just before initiating “sample preparation”, place MgCl_2 , 10X PCR buffer, BSA, dNTPs, unlabeled “EB1” and “EB2” primers, and SYBR Green I on a 48-position microcentrifuge rack in order to thaw these reagents.

IMPORTANT: In order to ensure accuracy and to minimize contamination, focus on the task and avoid unnecessary conversations.

F. Standard Curve Preparation

1. Retrieve standard DNA from the freezer labeled 1600 pg/ μL and record lot #.
2. Ensure that the contents of the 1600 pg/ μL standard DNA tube are thawed and removed from the cap, by centrifuging the tube.
3. Label tubes as follows: 400, 100, 25, 6.25, 1.56, 0.39, and NTC (no template control or 0 pg/ μL).
4. Add 15 μL of irradiated water to tubes 400, 100, 25, 6.25, 1.56, 0.39, and the NTC. Pipet tips do not need to be changed to add water to empty tubes. Close all caps.

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5. 0.25 Serial dilution

In order to mix each dilution thoroughly, either pipet the dilution up and down several times or vortex each dilution and subsequently centrifuge the tube at no more than 3000 rpm for 3 seconds.

- a. Open only two consecutive standard DNA tubes at once starting with the 1600 and the 400 pg/μL tubes. (Do not open the 0 pg/μL tube for this dilution.)
- b. Mix the DNA solution in the 1600 pg/μL. Take 5 μL of standard DNA at 1600 pg/μL and add to the 400 pg/μL tube, and thoroughly mix the contents.
- c. With a new pipet tip, take 5 μL of standard DNA at 400 pg/μL and add to the 100 pg/μL tube, and thoroughly mix the contents.
- d. With a new pipet tip, take 5 μL of standard DNA at 100 pg/μL and add to the 25 pg/μL tube, and thoroughly mix the contents.
- e. With a new pipet tip, take 5 μL of standard DNA at 25 pg/μL and add to the 6.25 pg/μL tube, and thoroughly mix the contents.
- f. With a new pipet tip, take 5 μL of standard DNA at 6.25 pg/μL and add to the 1.56 pg/μL tube, and thoroughly mix the contents.
- g. With a new pipet tip, take 5 μL of standard DNA at 1.56 pg/μL and add to the 0.39 pg/μL tube, and thoroughly mix the contents.
- h. Do not add anything to the NTC tube.

G. Sample Preparation

1. Remove calibrator: 1500 pg/μL from freezer and record lot number.
 - a. Vortex the calibrator thoroughly and centrifuge the tube at 3000 rpm for approximately 3 seconds.
 - b. Make three 0.166 dilution (1/6) of the calibrator with 4 μL of the calibrator and 20 μL of irradiated water.
2. Refer to Table 1 for sample submission guidelines.
3. Vortex all samples including the standards, NTC, calibrator, and the dilution and/or extract tubes.
4. Centrifuge all samples briefly for 3 seconds at no greater than 3000 rpm; this will prevent the DNA from aggregating at the bottom of the tube.

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5. **Witness Step:**

Arrange samples in order according to the sample sheet in a 96 well rack.

- a. Place samples in exactly the same place on the rack as they will appear vertically positioned in the rotor. (The microcentrifuge rack positions A1-H1, A2-H2, A3-H3, A4-H4, A5-H5, A6-H6, A7-H8, and A9-H9. In the newer rotors, the Rotor positions are 1-72. Therefore, rotor position 1 is equivalent to A1 on the rack, B1=2, C1=3, etc. In the older rotors, the rotor positions are A1-A8, B1-B8, C1-C8, D1-D8, E1-E8, F1-F8, G1-G8, H1-H8, and I1-I8. Therefore, A1 for the rotor is equivalent to A1 on the rack, B1=A2, C1=A3, D1=A4, E1=A5, F1=A6, G1=A7, H1=A8, and I1=A9.)
- b. Specifically, standard 1600a will reside in well 1 in the rotor (or A1 in the older rotors) and A1 in the rack. Standard 1600b will reside in 2 on the rotor (or A2 in the older rotors). However, well B1 will be empty, since one will take the duplicate 1600 standard from the first tube. Rotor position 3 (or A3 in older rotors) corresponds to rack well C1, standard 400a, and rotor position 4 (or A4 in older rotors), represents standard 400b in rack well D1 which is empty.
- c. Only the standards 1600 pg/ μ L – 0.39 pg/ μ L are duplicated. The no template control (0 pg/ μ L), the calibrator, and the samples are not duplicated.
- d. In brief, the standards are vertically placed in positions A1 through B4 in a 96 well rack.
- e. The no template control resides in B7, and the calibrator in B8. Samples are located in rotor wells 17-72 and rack positions A3-H9.
- f. Label the top of the sample tubes with rotor well identifier to minimize pipetting errors and sample mix ups.
- g. Have a witness confirm the sample locations.

H. **Master Mix preparation**

1. Prepare a 1/100 dilution of SYBR Green I.
Take 2 μ L of SYBR Green I in 198 μ L of irradiated water, vortex, and tap the tube on the bench to consolidate the reagent at the bottom of the tube.

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2. Mix each reagent before adding.
 - a. After each reagent has thawed, vortex each reagent, with the exception of TAQ GOLD.
 - b. Centrifuge reagents in the table top centrifuge at 3000rpm for approximately 3 seconds.
3. Add each reagent in the order as it appears on the worksheet. Thoroughly mix each tube reagent by pipetting up and down, or vortexing briefly. If vortexing, afterwards tap the tube on the bench to prevent the reagent from being trapped in the cap.
4. For total reagent volumes above 20 μ L, use a P200 even for multiple dispenses as opposed to one dispense with a P1000. To ensure accurate pipetting, aspirate and dispense the reagent as specified on the run sheet.
5. After adding each reagent, check that it has been added on the quantitation sheet, and place the reagent back in the Nalgene cooler, or for water and DMSO, in the opposite corner of the 48 well microcentrifuge rack.
6. Thoroughly mix the master mix by vortexing. Tap the tube on the bench to prevent the reagent from being trapped in the cap and/or centrifuge briefly for approximately 3 seconds.
7. Add 23 μ L of master mix to the appropriate number of Rotorgene tubes. Fill tubes in a vertical fashion (positions 1-16 or A1 to A8, and B1-B8 in older rotors). After adding master mix to 16 tubes, re-vortex the master mix and ensure all of the master mix is consolidated by tapping the tube on the bench and centrifuging briefly for approximately 3 seconds. Use a new pipette tip.
8. In order to track additions, it may be helpful to recite each well position to be oneself.

See below for reagent concentrations, the spreadsheet will calculate amounts for n+10% samples and will display rounded values for pipetting.

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TABLE 2:

Reagent	Concentration	μL [#] for 1 Rx
Irradiated GIBCO™ ULTRA PURE™ distilled water		8.3 (8.26)
10X PCR Buffer	10mM Tris/ 50mM KCL	2.5
25 mM MgCl₂	275 μM	2.8 (2.75)
5 mg/mL BSA	0.525μg/μL	4.0
2.5 mM dNTPs	200 μM each	2.0
DMSO	8%	2.0 (1.96)
1/100 dilution of 10,000X SYBR Green I	100X	0.3 (0.28)
20 pmol/μL Primer EB1	0.4 μM	0.5
20 pmol/μL Primer EB2	0.4 μM	0.5
5U/μL ABI Taq Gold	1.25U	0.3 (0.25)
Total volume		23.00

[#]The spreadsheet calculates the values using two significant figures. However, for the purposes of manual addition, only one significant digit is shown.

I. Sample Addition

1. In order to avoid the creation of aerosols, thoroughly mix the contents of each tube by pipetting up and down repeatedly.
2. Add 2 μL of each sample, including the standards, NTC, the calibrator dilution, and the sample dilutions and/or extracts, to each tube with master mix.
 - a. If necessary, in order to conserve sample, only 1 μL of sample may be measured. Note this in the sample name, and double the resultant value to accurately reflect the sample's concentration per microliter.
 - b. It is helpful to place the sample extract tube in its cryobox or another rack directly after its addition.
 - c. Every four reaction tubes, place caps on the tubes. (The caps are attached in sets of four.)

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- d. Number the first cap in every set of four as they will appear in the rotor. (1 for 1, 2 for 2, etc. For the older rotors, 1 for A1, 5 for A5, 9 for B1 etc.) **DO NOT** label the tube itself, as this may interfere with fluorescent detection.
- e. Open the machine. Remove the circular rotor from the instrument by either pressing in the middle silver stem in the RG6000 or unscrewing the center piece in the RG3000. Remove either the silver clip from the RG6000 rotor or the silver ring from the RG3000 rotor. Add tubes to the rotor. Ensure that tube 1 is in position 1, etc. or in older rotors, 1 is in position A1 etc.
- f. Ensure that all positions on the rotor are filled (using blanks if necessary).
- g. In the RG6000, add the silver clip to the rotor, lock into the Rotorgene, and close machine. In the RG3000, add the silver ring and screw the rotor into the Rotorgene, locking the rotor in place. Ensure the silver ring is in place and sitting securely in the rotor on all sides. Close machine.

J. Software Operation

1. Open excel and the relevant sample sheet to the sheet with the sample names, then collapse the window.
2. Open Rotorgene 6 software on the desktop.
3. Click File, New, Casework, and click “new”
4. In the wizard
 - a. Ensure that the “Rotorgene 72 well rotor” is highlighted
 - b. Make sure that the box next to “locking ring attached”, is checked.
 - c. Click “Next.”
 - d. Type initials for Operator and add any notes (extraction date/time)
 - e. Reaction volume should be “25 µL”
 - f. Sample layout should be “A1, A2, A3,...”
 - g. Click “Next.”

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- h. In the RG3000s, click “Calibrate”. In the RG6000, click “gain optimisation”.
 - i. “Perform Calibration before 1st acquisition”
 - ii. Click on “calibrate acquiring” (RG3000) or “optimize acquiring” (RG6000).
 - iii. “This will remove your existing setting for auto gain calibration?” The window appears, click YES. A green gain window will open. Click “ok”, then “close”.
 - iv. Note selecting “calibrate all” will attempt to calibrate for all channels known by the software whereas “calibrate acquiring” will instead only calibrate those that have been used in the thermal profile defined in the run such as FAM or Green.
 - v. Click next in wizard and “start run”.
5. “Save as” the RG#, date and time (for example, “RG1Q112904.0200” for a run on RG1 on Nov 29, 2004 at 2:00) in Log Archive folder.
6. Sample sheet window
 - a. Expand the excel sample sheet window. Copy the sample names.
 - b. Paste sample names in the appropriate rows in the Rotorgene sample window by right clicking and selecting paste.
 - c. Settings:
 - i. Given concentration format: 123,456.78 unit pg/μL
 - ii. Type category
 1. Standards: std
 2. Zero standard: NTC
 3. Samples and calibrator: unk
 - iii. In all wells with standard, calibrator or sample, select “YES”
You may copy and paste selections by right clicking.
 - d. Hit “Finish”

See below for cycling parameters that should not be changed:

95°C	10 min	35 cycles
94°C	15 sec	
68°C	60 sec	
72°C	30 sec	
72°C	15 sec	

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7. Enter run information in the Rotorgene log book.
8. The run will approximately require 1 hour and 40 minutes for completion.
 - a. If you are leaving for the day, the machine needs no further assistance.
 - b. If you will remain upon completion of the run, and no more runs will be conducted that business day, turn off or log off the computer when the run completes.
9. Following the initial heating to activate the TAQ and the gain calibration, the raw data will appear on the screen. With this information, one can monitor the progress of the run. Fluorescence for the highest standard should be apparent from ~ cycle 15.
10. Previous run files may be examined while the computer is collecting data.
 - a. Collapse the window.
 - b. Double click on the Rotorgene icon on the desktop.
 - c. The computer will prompt that another version of the software is running and ask if you want to run an analysis version only. Click yes.

K. Clean Up

1. Wipe entire work area surface and pipets with bleach followed by water and EtOH.
2. Soak and/or clean racks, pipets, cap openers, and other equipment that would not be adequately cleaned with the stratalinker with 10% bleach, 70% ETOH, and water. (Ensure that bleach is completely removed from the equipment.)
3. If necessary, place fresh microcentrifuge tubes in the stratalinker and apply ultraviolet radiation for the 30 minutes.
4. Turn on the UV lamp in the hood for ten minutes. Do not forget to turn off the light.
5. Return water, dNTPs, MgCl₂, 10X PCR buffer, BSA, DMSO, EBI primer, EB2 primer, TAQ GOLD and water tubes with any remaining reagents to storage.
6. Dispose of all dilution tubes of the standard, calibrator, and SYBR Green I. Sample aliquots may be stored until assay success is confirmed.

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L. Sample and Data Storage

1. Store extracts in a cryobox in the DNA refrigerator. For LCN, the extracts should be stored in the DNA refrigerator in the pre-amp room in the designated area.
2. Ensure that the final Rotorgene sheet is stored on the network in the folder labeled “RG sheets” and that the data from the assay is in the folder labeled “RG data” under the appropriate Rotorgene folder.
3. To transfer over the Rotorgene data to the network:
 - a. After the run is done, save and exit out of the Rotorgene software.
 - b. In the Log Archive, go to the appropriate run folder.
 - c. Copy the run onto a flash drive and transfer the run into the appropriate Rotorgene folder under the “RG data” folder on the network. (To remove the flash drive from the computer, click on the green arrow icon on the lower right corner of the computer screen. Click “Safely remove USB Mass Storage Device”.)
4. Pass the assay and sample sheets to the rotation supervisor for review.

M. Analysis

1. Analysis may be performed on the instrument computer or any computer that has access to the software.
2. Open Rotorgene software on the desktop. If the computer is not connected to an instrument, when the software indicates that the computer cannot connect to the instrument on serial port COM1, select “run in virtual mode”.
3. Click “Open” and click on the run to be analyzed in the Log Archive folder

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4. Click “Analysis” on the toolbar.
 - a. Select **“Quantitation”, “Show”**.
 - i. Three windows will open with the standard curve, the samples, and fluorescence.
 - ii. If a “Calculate Automatic Threshold” window will show up, click ok.
 - iii. Ensure that **“dynamic tube”** and **“slope correct”** are selected on the tool bar.
 - iv. Select the tab **“more settings”**.
 - 1) Ensure that the NTC threshold is set to 10%.
 - a) This permits the exclusion of samples or NTCs, which have a slight drift upwards, due to probe degradation or other non PCR effects. All samples with a change below the NTC threshold of 10% will not be reported in the quantitation screen. The percentage is relative to the largest maximum change found in any tube.
 - b) For example, if you have one sample which began at a background of 2 FI and went to 47 FI, then 45 FI represents 100%. An NTC threshold of 10% would consider as noise any sample with a reaction less than 4.5 FI.
 - 2) The box under the **“reaction efficiency threshold”** should NOT be selected however.
 - a) With this alternative mechanism to exclude noise samples from analysis, all samples will be excluded if they do not have a reaction efficiency of at least the specified this level. A level of 0% indicates that, during the exponential phase, no reaction took place. 100% indicates that a completely efficient reaction took place during the exponential phase. Negative percentages indicate that during what has been guessed as the exponential phase, the fluorescent signal declined.
 - b) Any sample with a genuine reaction will have some visible exponential phase with some growth. Setting this value higher than 0% will exclude some samples with inefficient, but perceptible growth, whereas setting below 0% will display samples whose reading decreased during the exponential phase, and which should clearly be excluded.
 - 3) Click “okay”
 - v. If any of the settings need to be corrected, “auto find threshold” must be performed again. (“Auto find threshold” can be found in the lower right corner of the screen if the “Quantitation Analysis” graph is selected.)

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- b. Check if any sample curve crosses the threshold at an early cycle due to background fluorescence. The sample in question would have no value, but the normalized data would display a curve that crosses the threshold both at an early and at a later cycle (Figure 1A).
 - i. Dynamic tube normalization uses the second derivative of each sample trace to determine a starting point for each sample. The second derivative measures the rate of the rate of change. In other words, the increase in fluorescence from cycle to cycle is averaged from cycle 1 up to the where the fluorescence “takes off” or the starting cycle number for each sample. This method gives the most precise quantitation results.
 - ii. In order to avoid disabling the dynamic tube normalization setting, move the threshold to the right, ignoring the first few cycles, so that the sample does not cross the threshold. This can be achieved by the following:
 - 1) In the normalized data windowpane, on the lower right side, under CT calculation, change the number for “Eliminate Cycles before:” from 0 to 1-5. Chose the smallest number where the threshold does not cross the data curve in question (Figure 1B).
 - 2) Alternatively, select the grid immediately to the right of “Eliminate cycles before”. This allows manual manipulation of the starting cycle number of the threshold.
 - 3) Reanalyze the data by selecting “auto find threshold”.
 - c. One may also manually manipulate the vertical position of the threshold on the standard curves.
 - i. Select the grid to the right of the threshold value and then click on the red threshold line and adjust the line. Moving this line vertically will make the threshold cross the standards’ curves at different cycles and thus will change the efficiency, Ct, and sample values.
 - ii. Position the line to optimize the distance between the Ct values of the standards and thus the calibrator values, while maintaining a passing efficiency value.
5. Save experiment as RG#, “Q”, date and time (“RG1Q052506.1100” for example) in the log archive folder.

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N. Report

- On the “Quant results” screen, (by right clicking the table heading with the mouse and un-checking certain columns) only pick the following columns: No., Name, Type, Ct., and Calc.Conc.
- Select “Reports” from toolbar
 - Select “Quantitation, cycling A FAM/SYBR”
 - Select “full report” double click
 - Generate report
 - Print at 95%.
- Submit reaction sheet and full report paperwork to supervisor for review.

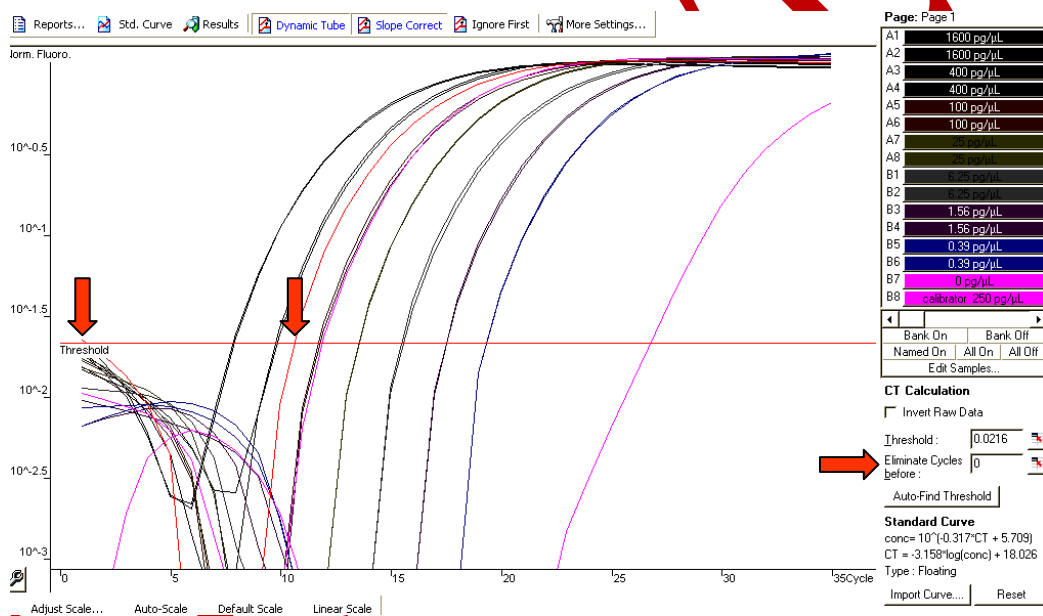


Figure 1A: Normalization with the dynamic tube method usually does not required one to eliminate early cycles. However, the sample depicted by the red curve above crosses the threshold during the first and the ninth cycle yielding a negative value.

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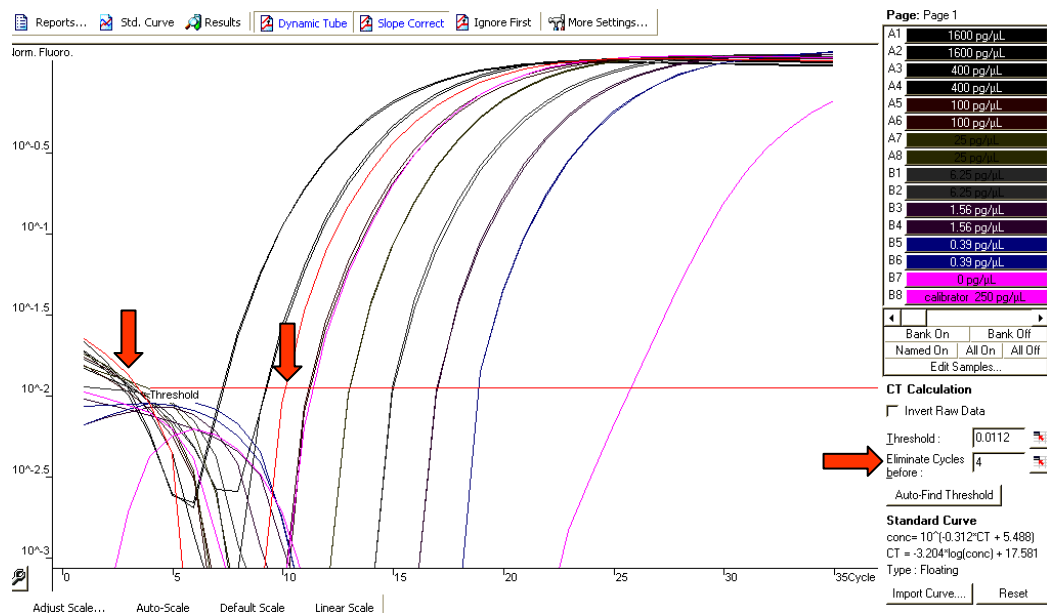


Figure 1B: Eliminating the first four cycles prevented the sample depicted in red from crossing the threshold at an early cycle. Therefore, the value at which the sample crossed the threshold, cycle nine was employed to determine the quantitative value.

O. Value Calculation by Software

1. Fluorescence values are recorded during every cycle and represent the amount of product amplified to that point in the amplification reaction.
2. The more template present at the beginning of the amplification reaction, the less cycles that will be required to obtain sufficient fluorescence signal above background. This point is defined as the Ct and will occur during the exponential phase of amplification.
3. The rotorgene software scans through all possible threshold levels until the best fit is found for the standard curve.
4. The R value is a number between -1 and 1 which defines how well a line of best fit or a least squares line describes the relationship between two variables. An R-value of zero implies no relationship, whereas an R-value of -1 or 1 indicates that the data values fall directly on the line of best fit.

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5. The following are calculated :
 - a. The slope of a reaction: change in Ct/change in log input, opt -3.322
 - b. Reaction efficiency = $[10^{(-1/\text{slope})}] - 1$, optimum 1
A 100% efficient reaction results in a doubling of the amplification product each cycle.
 - c. exponential amplification = $10^{(-1/\text{slope})}$, optimum 2

P. Assay Interpretation

Standards and Controls:

1. Check the raw data for cycling. (If the raw data graph is not seen, click on "Cycling A.Green" in the tool bar and then "Arrange".) If the fluorescence is below 80 RFUs, yet the reaction efficiency is acceptable (see 5), determine if the SYBR Green I was thawed more than once. If not, notify QC in order to test stock. The assay still passes as long as conditions 2b and 3 are fulfilled.
2. Confirm that the following settings are correct:
 - a. standard curve imported "no"
 - b. Start normalizing from cycle "1"
 - c. noise slope correction "yes"
 - d. reaction efficiency threshold "disabled"
 - e. normalisation method "dynamic tube normalisation"
 - f. digital filter "light"
 - g. no template control threshold "10%"
3. Slope optimum: -3.322
4. R^2 value optimum: 0.999
5. **Reaction efficiencies should range from 0.88 to 1.15.**
 - a. **Reaction efficiencies above 1.15 are too high and the run fails.**
 - b. **Reaction efficiencies below 0.88 are too low and the run fails.**
 - c. **Efficiencies are rounded down. (For example, 0.879 fails.)**
6. **Two of the three calibrator values must be between 400 pg/μL and 100 pg/μL.**
 - a. This assay has a potential inherent error of a factor of 0.3.
 - b. Therefore, the expected value for the calibrator is between 175 pg/μL and 325 pg/μL.

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7. **Non template controls or zero standards should be < 0.1 pg/μL.** When the curve of the NTC does not cross the threshold, no value will appear in the concentration column. Assume that the value is zero. However, confirm that the curve from the raw data does curve slightly and is not a flat line. A flat line indicates that inhibition has occurred (refer to Sample Interpretation section 5b.) There will always be some background amplification at the late cycles even the NTC.
8. **The difference between the average Ct values of each consecutive duplicate standard concentration should be approximately two cycles.**
9. **At least one of each duplicate standard concentration should be apparent (“clicked on”). (If #10 is exercised, at least one of each duplicate standard concentration should be apparent for 5 of the 7 remaining standards.)** If one duplicate of a standard does not yield the expected Ct value, but the other duplicate is within the expected range, the aberrant standard may be excluded from the standard curve calculation. Unselect the sample on the right side of the screen, and reanalyze.
10. Similarly, if both replicates of a standard are not within the expected range, they may both be excluded from the standard curve calculation, and if all the other parameters of the assay are satisfactory, the assay passes. **However, no more than two standard pairs may be absent.**
11. Record the reaction efficiency, calibrator and non-template control values, and fail the assay if these values or the standards are unacceptable. The reviewer must sign and date the sheet as well.
12. For LCN, complete an Assay Resolution sheet if necessary and identify the problem, the assay name, and the samples affected, and select a resolution from the drop down menu. Typically, the samples will need to be re-quantitated. For LCN samples, in order to preserve sample, if the quantitation assay fails twice, proceed to amplification without a third quantitation.
13. Initiate retesting of all samples in a failed run. Although a quantitation assay may fail, the resultant values may be used to estimate the need for further dilutions for the re-quantitation assay.

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TABLE 3:

Required Settings		Required Results	
Parameter	Value	Parameter	Value
Start normalizing from cycle	1*	NTC	< 0.10 pg/μL
Noise slope correction	yes	Calibrator	100 to 400 pg/μL
Reaction Efficiency threshold	Disabled	Reaction Efficiency	0.88 to 1.15
Normalization Method	Dynamic tube Normalization	Ct values of standards	~2 cycles between each concentration
Digital Filter	Light	Standards analyzed	No more than 2 pairs may be absent
No template control threshold	10%	Samples	<1000 pg/μL or dilute and re-quantitate
		Sample Notes	“*” if background fluorescence “Δ” if inhibited

* May change if a sample curve crosses the threshold early (refer to Section M.4.b.ii. of this section).

Sample Interpretation (Tables 3 and 4):

1. Samples that are 1000 pg/μL and above should be requantitated at a 1:100 or a 1:1000 dilution.
2. For LCN samples, if the extraction negative is > 0.2 pg/μL the sample set must be re-extracted prior to LCN amplification. A concentration of 0.2 pg/μL would equate to 1 pg of DNA amplified with Identifiler™ for 31 cycles, which would produce ample alleles to fail the amplification.
3. Similarly, amplification of 1 pg with Identifiler™ reagents for 28 cycles may generate 1 allele which would fail the amplification. Thus, if the extraction negative is > 0.2 pg/μL the sample set must be re-extracted prior to amplification.
4. Since the Cofiler™/Profiler Plus™ 28 cycle systems are less sensitive, the thresholds for amplification are higher. If the extraction negative is > 1 pg/μL or 20 pg/amp for Cofiler/Profiler Plus it will need to be re-quantitated (refer to Table 4 below).

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5. When the curve of the extraction negative does not cross the threshold, no value will appear in the concentration column. Assume that the value is zero.

TABLE 4:

Amplification System	Sensitivity of Amplification	Extraction Negative Control Threshold
Cofiler™/Profiler Plus™/YM1	20 pg	1.00 pg/μL in 20 μL
Identifler™ 28 cycles	1 pg	0.20 pg/μL in 5 μL
Identifler™ 31 cycles	1 pg	0.20 pg/μL in 5 μL

6. If the no template control is > 0.1 pg/μL, LCN samples may be amplified since there may not be sufficient sample to retest. However, HCN samples must be requantitated.
7. If a sample appears to be inhibited since, the curve initially increases and then plateaus (see Figure 2), purify the sample with a Microcon® 100 and amplify or re-quantify if necessary. (If the raw data graph is not seen, click on “Cycling A.Green” in the tool bar and then “Arrange”.)
- Figure 2A displays samples that inherently fluoresce. Indicate such samples with an asterisk, “*”, on the sample sheet in the Notes column.
 - For other types of inhibition and/or quenching, the curves will appear as the other curves but then plateau earlier (Figure 2B). Note this type of inhibition with a triangle, “Δ”. Although purification of the samples in Figure 2B may enhance fluorescence in this assay, since sufficient DNA is present for amplification, the samples produced DNA STR profiles (data not shown). The important factor is whether the samples increase in fluorescence by at least ten fold.

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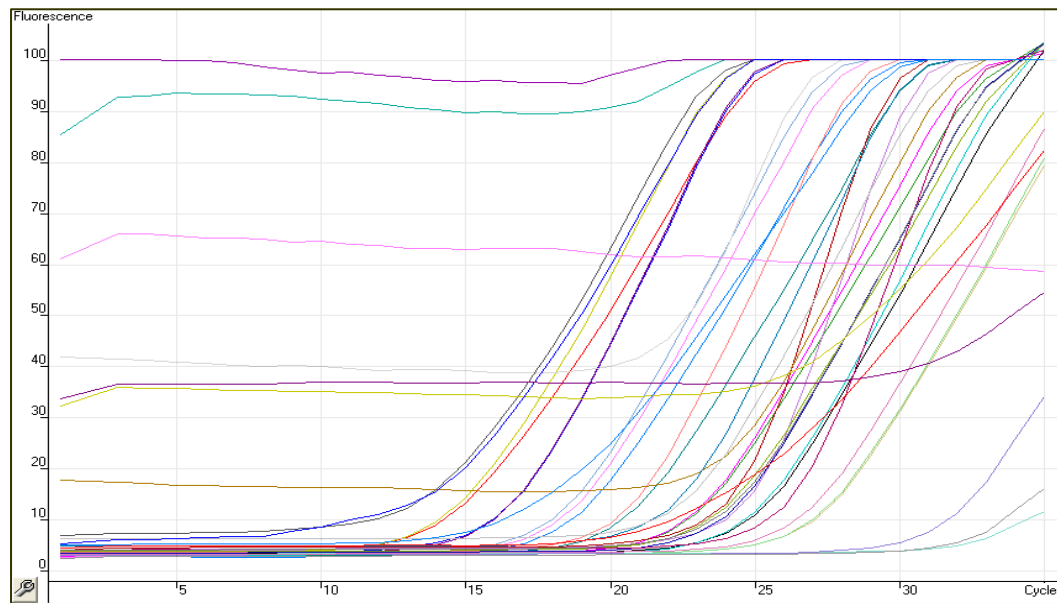


Figure 2A: The plateau lines are examples for inhibited samples that have inherent fluorescence. The slopes starting at cycle 15 are the expected DNA value increase.

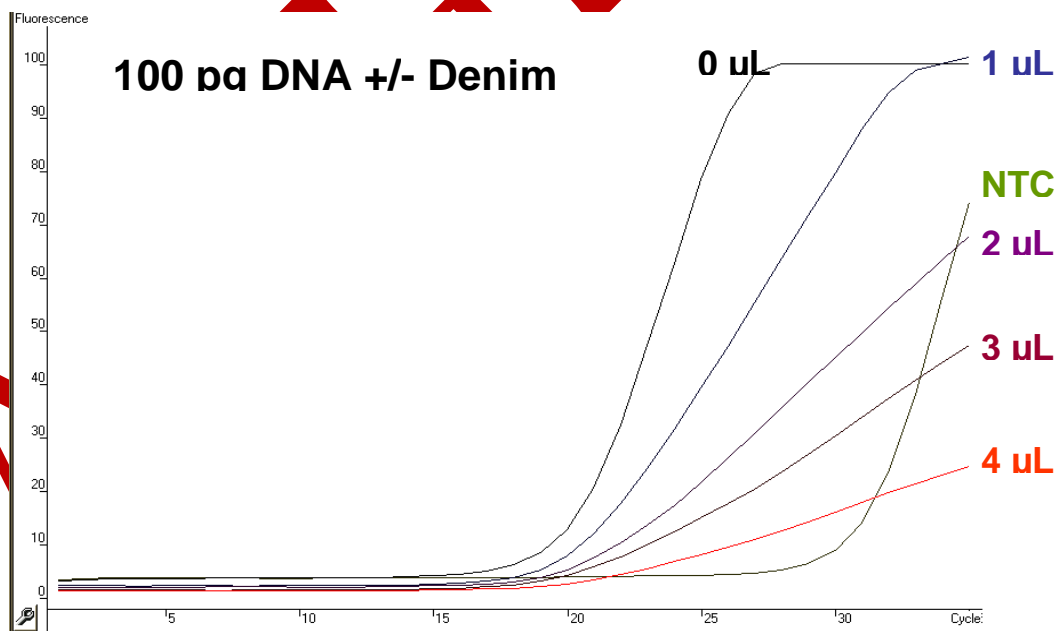


Figure 2B: 100 pg of DNA was measured with 0 to 4 µL of denim dye. The addition of denim dye to the samples reduced the amount of recorded fluorescence. However, DNA was still amplified, as evidenced by the increase in fluorescence over time.

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8. If the baseline curve of a sample's raw data is below that of the standards in early cycles, this may suggest the presence of a substance that is quenching fluorescence (i.e., dyes, such as denim, or hemoglobin).
 - a. This can be confirmed by using the cursor to draw a box around the baseline of the samples in the "Raw Channel (Cycling A. Green)" window, selecting "Zoom" from the pop-up menu and inspecting the RFUs of the samples in the early cycles relative to that of the standards. Refer to Figure 3.

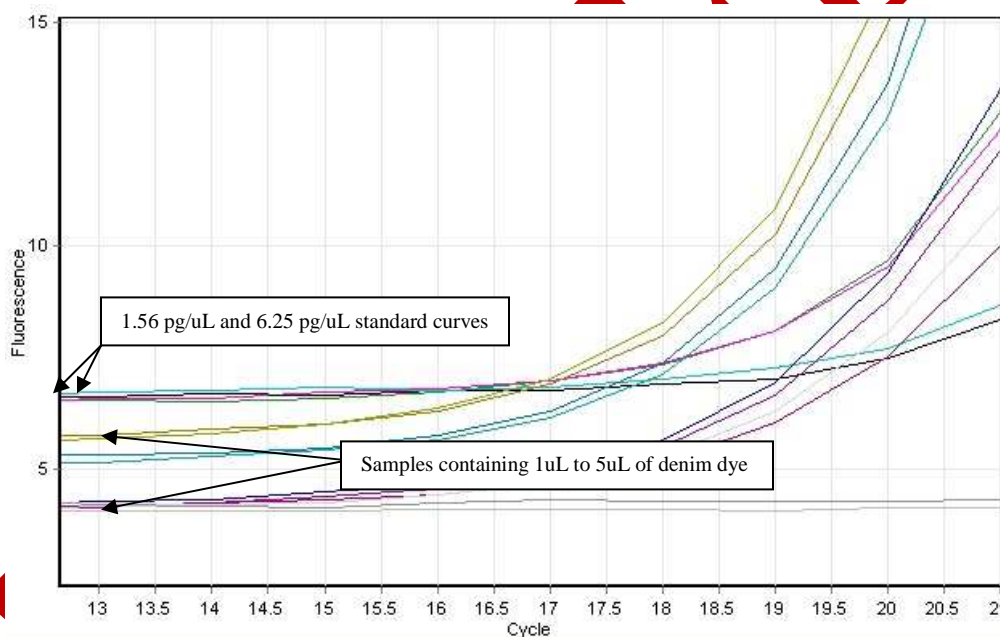


Figure 3: Raw data for samples containing a quenching dye. For samples containing 1uL to 5uL of denim dye, the more dye added to a sample, the lower the RFUs recorded in the early cycles. The RFUs of all samples containing dye showed baseline fluorescence approximately 1-2 RFUs below the standards depending on the amount of dye added.

- b. If, as in Figure 3, the RFUs of the samples fall below those of the standards, a substance could be present that is quenching the fluorescence of the SYBR Green I dye. Non-sigmoidal curves may or may not also be associated with these samples depending on the amount of the quenching substance in the sample. Additional purification steps may be necessary for these samples as determined by the analyst or rotation supervisor.

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9. Proceed to amplification and/or purification/concentration for all samples with values according to Table 4. If a sample requires purification or purification and concentration re-quantitate following purification. However, samples simply requiring concentration may be amplified directly.

TABLE 5:

	Cofiler™/ Profiler Plus™	Identifiler™ 28 cycles	Identifiler™ 31 cycles
Minimum Desired Template	150.00 pg	100.00 pg	^20.00 pg
Template volume for amp	20 µL	5 µL	5 µL
Minimum Sample Concentration in 200 µL	30 pg/µL	20 pg/µL	^30 pg/µL
Minimum Sample Concentration in 200 µL prior to Microconning* to 50 µL	1.87 pg/µL	5 pg/µL	N/A
Minimum Sample Concentration in 200 µL prior to Microconning** to 20 µL	N/A	3 pg/µL	0.40 to ^0.10 pg/µL
For LCN samples: Minimum Sample Concentration in 20 µL	30.00 pg/µL	20.00 pg/µL	4.00 to ^1.00 pg/µL

* Sample concentration **prior** to processing with a Microcon 100 and elution to 50 µL

** Sample concentration **prior** to processing with a Microcon 100 and elution to 20 µL

^ Samples with less than 20 pg per amplification may be amplified upon referral with the LCN supervisor

10. The neat and the 1/100 dilutions of the samples should differ by a factor of two. For example, if the neat value is 200, the 1/100 value should be between 1.0 and 4.0 and optimally be 2.0. If the dilutions are not within a factor of 2.5, the samples should be re-quantitated with the following exceptions:
- When the more concentrated dilution approaches or is above 1000 pg/µL, the amplification may be saturated and therefore, the dilutions may not be within the specified values. In this instance, the 1/100 value should be used for estimation of the DNA input for amplification.

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- b. When quantitation values from both dilutions are close, the more concentrated dilution, the neat dilution for example, may be inhibited and therefore generates a small value. This would be indicated in the raw data as described above. If when preparing a sample for amplification, the 1/100 dilution would be used and showed no inhibition with quantitation, then use this dilution. However, if the neat would be used to prepare a sample for amplification, the sample may require purification. For HCN samples, this purification will occur at the analyst's discretion. If the 1/10 dilution would be used for submission for amplification, the degree to which the neat dilution was inhibited with the neat dilution will influence whether the sample should be amplified directly or purified.
- c. As a general guide, if the curve of a sample shows background fluorescence only and is not inhibited, the sample may be submitted for amplification using the 1/100 dilution value. This scenario is sometimes observed with semen samples. However, as stated above, if the raw data curve indicates inhibition such as that shown in Figure 2B the sample may require purification.

Q. Creating a Rotorgene Summary Page

1. Open the Rotorgene summary sheet excel file for the Rotorgene run being analyzed and reviewed. The run will be saved with the run name in the folder for that instrument, such as RG3Q011707.1100 saved in the RG3 folder. Go to the "Copied Values from Rotorgene" tab.
2. On the Rotorgene Software (main screen after analysis), go to the "Quant. Results - Cycling A..." table (lower left window).
3. Maximize the screen. By right-clicking the table heading with the mouse and unchecking certain columns, eliminate all columns except the following:
No
Name
Ct
Calc. Conc.
4. Select all remaining cells (left click and drag across all column headings until all cells are highlighted blue). Then, right-click mouse and select copy.
5. In the Rotorgene summary sheet excel file in the "Copied values from Rotorgene" sheet, make sure well **B1** is selected. Right click on well B1 and paste special as text. In row 1, the column headings should be visible.

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6. If the extraction negative does not cross the threshold, the sample is not inhibited, and there is no value, ensure a value of zero is entered into the calculated concentration column.
7. Fill in tube labels for respective cases in column A if applicable. (Enter “-” for standards, negative controls and calibrators.) The tube labels may also be copied and pasted special as “values only” from the “sample sheet” or typed in manually. If using “copy” and “paste special”, make sure the right tube labels correspond to the right cases, especially if certain standards were eliminated during analysis.
8. Enter the IA initials and target dates for the respective cases in column F and G, if applicable. The IAs and the target dates may also be copied and pasted special as “values only” from the “sample sheet” or typed in manually. If using “copy” and “paste special”, make sure the right tube labels correspond to the right cases.
9. Go to the “Summary Sheet” tab, and check to make sure all sample names fit in respective cells.
10. The RG summary sheet will automatically place an “RQ” next to samples with quant values greater than 1000pg/uL. Inspect these samples to ensure that a requant is in order. If the dilution can be used, right click on the “Comments” cell for that sample and “clear contents”.
11. Schedule samples for re-quantitation if needed, by placing an “RQ” in the comments section next to those samples. Any sample with a lowest dilution quant value of greater than 1000 pg/uL should be re-quantified. Also, any sample pair with values for the neat and diluted samples that do not correlate should also be re-quanted (see #7 in this section for re-quantitation guidelines). Note that although a quantitation assay may fail, the resultant values may be used to estimate the need for further dilutions for the re-quantitation assay.
12. Save.
13. Minimize the summary sheet and inspect the raw data for inhibition and background fluorescence. Inhibited and/or fluorescent samples should be noted in the “Comments” column with a “Δ” and/or a “*”, respectively. The “Δ” may be copy and pasted from the footnote section.
14. For LCN casework, the supervisor may indicate whether a sample requires purification by inserting a “P” in the comments section.

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15. Print the summary sheet pages. If the sample batch size is such that all samples are included on page 1, only page 1 requires printing.
16. While on the summary page, hit the Ctrl + q keys to automatically import all samples requiring requantitation. The IA, sample description, original RG results and original RG run will be automatically imported. Manually fill in the extraction date and time, the cryobox in which to find the samples and the dilutions at which to rerun the samples or purification recommendations. This sheet may then be maintained electronically or printed out and used as a hard copy.
17. The “post-quantitation resolution sheet” may be saved as an independent sheet for subsequent additions by right-clicking the “requant” tab and selecting “move or copy”. In this window, select “(new book)” in the “to book” window and check “create a copy”. Click “OK”. Go to File – Save As and save into the appropriate folder with the start date of the sheet. When a sheet is full archive the file in the associated Archive folder. Samples may be manually added to these sheets or copied and pasted special from requant sheets at the end of each Rotorgene run.
18. On the hard copy of the summary sheet and the assay sheet, note the reaction efficiency, the calibrator values and the NTC values at the top of the summary sheet and the bottom of the assay sheet.
19. The reviewer must also sign and date the summary and assay sheets and indicate whether the assay has passed or failed.

R. File Protection and Paperwork Distribution

1. Following assay review, the reviewer must protect the electronic file by designating it as a “read only” file.
 - a. In windows explorer, locate the relevant file folder which should be found in the rotorgene folder within the folder for the instrument used.
 - b. Right click on this file and select properties. Under attributes, chose “read only” and select “apply” then “ok”.
2. File the full assay report from the Rotorgene and the sample and reagent sheets. Only distribute the Rotorgene summary sheet to analysts.

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References:

Nicklas, J. A., Buel, E. Development of an *Alu*-based, Real-Time PCR Method for Quantitation of Human DNA in Forensic Samples

Nicklas, J. A., Buel, E. Development of an *Alu*-based, QSY 7-Labeled Primer PCR Method for Quantitation of Human DNA in Forensic Samples

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Revision History:

October 6, 2006 – This section was originally from the Protocols for High Sensitivity Testing manual. It was updated and clarified for use in HCN casework. See [Approval Form](#).

January 31, 2007 – Entire section edited for clarification for use in HCN casework. See [Approval Form](#).

June 1, 2007 – Edited to include Identifiler 28 for use in casework. See [Approval Form](#).

March 13, 2008 – Edited to change the amount of TAQ Gold and annealing time for RG procedure. See [Approval Form](#).

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A positive control, an amplification negative control, an extraction negative control, and a female negative control, if applicable, should be included with each batch of samples being amplified to demonstrate procedural integrity. The positive control is a control whose alleles are known to the analyst. Samples that were extracted together should all be amplified together, so that every sample is run parallel to the extraction negative control.

Do not forget to indicate on the DNA extract tracking worksheet that a sample was submitted.

The STR PCR reaction mix already contains BSA. **Do not add additional BSA.**

Follow the general PCR guidelines for handling the tubes and cleaning of the work surface. The following steps have to be performed in the appropriate dedicated areas. Evidence samples and exemplars should not be handled at the same time.

A. Preparing DNA aliquots for the amplification

1. Do not amplify samples in which insufficient DNA was detected by QuantiBlot (0 or $<0.15\text{ng}/20\mu\text{L}$). These samples may be amplified after a Microcon concentration step.
2. The target amounts to be amplified are 1 ng of DNA for the Profiler Plus and Cofiler multiplexes, 0.5ng for Powerplex16, and 2ng for the Y-STR systems. Due to the varying volumes of reaction mix and magnesium chloride for the different multiplex reactions, the DNA aliquots have to be adjusted accordingly to achieve a final volume of $50\mu\text{L}$, or $25\mu\text{L}$ respectively. Follow the appropriate tables when setting up the aliquots.

ATTENTION: Powerplex16 is currently only validated for exemplars and human remains. Do not use for evidence specimen.

3. For each sample to be amplified, label a new tube. Add DNA and TE^{-4} and as specified in Tables 1A to 3A.
4. Remember the following general rules:

If the heat and 1/10 QuantiBlot results are $\geq 10\text{ ng}/20\mu\text{L}$, dilute the sample 1/100 with TE^{-4} and re-quantitate. Multiply the diluted concentration by 100 to obtain the original sample concentration. Repeat the procedure if the diluted sample is still $\geq 10\text{ ng}/20\mu\text{L}$.

When a dilution is made to determine DNA concentration, it is preferable to calculate the DNA concentration in the undiluted DNA extract. Then amplify the undiluted DNA extract, not the dilution. *If possible always amplify the optimal target amount.*

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Powerplex 16 multiplex

Table 1A - Amount of DNA to be amplified for Powerplex 16

Quantiblot DNA Concentration (ng/20µL)	Target Volume (µL) to be amplified	TE ⁻⁴ (µL) for Target Volume	Range of Volumes (µL) which can be amplified (Corresponds to approx. 0.15 - 5 ng of DNA)
≥25	Dilute 1/10 and use the dilution for amplification		
12.5	1.6	18.4	0.3-8**
6.2	2.0	18.0	0.5-16**
5	3.2	16.8	0.6-20**
3.1	4.0	16.0	1.0 - 20**
2.5	6.6	13.4	1.2-20**
1.5	8.0	12.0	2.0 - 20**
1.25	16.0	4.0	2.4-20**
0.62	20	0	4.8-20**
0.31	20	0	9.6 -20**
0.15	20	0	19.2-20**
< 0.15	Do not amplify***	-	-

** Add TE⁻⁴ to a final volume of 20 µL.

*** This does not apply to Missing Persons/Unidentified Body cases. Tissue samples and reference samples may be amplified with <0.15 Qblot value. Submit 20ul for PowerPlex 16 amplification only.

Table 1B - Control samples for the Powerplex 16 multiplex

Sample	DNA	TE ⁻⁴
Promega Kit positive control	10 µL	10 µL
Extraction negative control	20 µL	---
Amplification negative control	---	20 µL

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Cofiler and Profiler Plus Kits

Table 2A - Amount of DNA to be Amplified for the Cofiler and Profiler Plus Kits

DNA Concentration (ng/20µL)	Target Volume (µL) to be amplified	TE ⁻⁴ (µL) for Target Volume	Range of Volumes (µL) which can be amplified
≥25	Dilute 1/10 and use the dilution for amplification		
12.5	1.6	18.4	0.3-8**
6.2	3.2	16.8	0.5-16**
5	4	16	0.6-20**
3.1	6.5	13.5	1.0 - 20**
2.5	8	12	1.2-20**
1.5	13.3	6.7	2.0 - 20**
1.25	16	4	2.4-20**
0.62	20	0	4.8-20**
0.31	20	0	9.6 -20**
0.15	20	0	19.2-20**
< 0.15	Do not amplify	-	-

** Add TE⁻⁴ to a final volume of 20 µL.

Table 2B - Control samples Cofiler and Profiler Plus Kits

Sample	DNA	TE ⁻⁴
ABI kit amplification positive control	20 µL	---
Extraction negative control	20 µL	---
Amplification negative control	---	20 µL

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Y STR multiplex YM1

The amplification of exemplars, sperm cell fractions of samples extracted by differential lysis and semen stains, where no epithelial cells were seen during the differential lysis, is based on the QuantiBlot results (see Table VI). Semen positive swabs taken from female individuals that were extracted using the non-differential semen extraction and the swab remains fractions of differential lysis samples are amplified using the amounts specified in Table 3B.

Table 3A - Amount of DNA to be amplified for the Y M1 based on QuantiBlot results.

QuantiBlot DNA Concentration (ng/20µL)	Target Volume (µL) to be amplified	TE ⁻⁴ (µL) for Target Volume	Range of Volumes (µL) which can be amplified (Corresponds to approx. 0.15 - 5 ng of DNA)
≥25	Dilute 1/10 and use the dilution for amplification		
12.5	3.2	22.8	0.3 - 8**
6.2	6.6	19.4	0.5 - 16**
5	8.0	18.0	0.6 - 20**
3.1	13.0	13.0	1.0 - 26**
2.5	16.0	10.0	1.2 - 26**
1.5	26.0	0	2.0 - 26**
1.25	26.0	0	2.4 - 26**
0.62	26.0	0	4.8 - 26**
0.31	26.0	0	9.7 - 26**
0.15	26.0	0	20 - 26**
< 0.15	Do not amplify	-	-

** Add TE⁻⁴ to a final volume of 26 µL.

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Table 3B - Amount of DNA extract from a non-differential semen extraction or from the swab/substrate remains fraction of a differential lysis sample to be amplified for YM1.**

P 30 result for the 2ng subtraction (Body cavity swabs)	P 30 result for the 0.05A units subtraction (Stains or penile swabs)	DNA Volume (μL) to be amplified	TE ⁻⁴ (μL)	Range of Volumes (μL) which can be amplified
ERR for the neat	ERR for the neat	5	21	1 - 26*
Between 1.1 - 3.0	Between 1.1 - 3.0	10	16	2 - 26*
> 0 - 1.0	> 0 - 1.0	26	0	5 - 26*

* Add TE⁻⁴ to a final volume of 26 μL.

** Use 10μL for samples where P30 was not determined because sperm cells were seen.

Table 3C - Amount of DNA extract to be amplified for Amylase positive samples.**

Type of item		DNA Target Volume (μL)	TE ⁻⁴ (μL)	Range (μL)
Vaginal swab	Initially try two amounts	10 26	16 0	1 - 26*
Anal or dried secretions swab	Based on Qblot result	see Table 3A.1		
Stain	Based on Qblot result			

* Add TE⁻⁴ to a final volume of 26 μL.

** Qblot does not reflect male DNA especially for vaginal swabs. Try more or less if negative.

Table 3D - Control samples Y STR multiplex YM1

Sample	DNA	TE ⁻⁴
in house amplification positive control	20 μL	6 μL
extraction negative control	20 μL	6 μL
amplification negative control	---	26 μL

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B. Amplification set-up

ATTENTION: Powerplex16 is currently only validated for exemplars and human remains. Do not use for evidence specimen.

1. For each system fill out the amplification worksheet and record the appropriate lot numbers.
2. Determine the number of samples to be amplified, including controls and label a PCR reaction mix tube for each sample.
3. Ensure that the solution is at the bottom of each PCR reaction mix tube by tapping the tube down onto a clean work surface or by centrifuging briefly. Label the caps of the PCR Reaction Mix tubes or use white labels. Open caps using the microcentrifuge tube de-capping tool or a new Kimwipe. **Avoid touching the inside surface of the tube caps.**
4. For Powerplex 16 the reaction mix has to be prepared fresh. Prepare enough for N+1 samples and mix before using:

Reagent	1 sample	10 samples	30 samples
Powerplex 16 10x primer mix	2.5 µL	25 µL	75 µL
Gold Star 10 x buffer	2.5 µL	25 µL	75 µL
AmphTaq Gold DNA Polymerase (5U/µL)	0.8 µL	8 µL	24 µL

5. According to the multiplex that is being amplified the following reagents have to be added to each tube:

	SYSTEM		
	Powerplex 16	Y STR YM1	Cofiler, Profiler Plus
Reagent	5.8 µL mastermix	4 µL of MgCl ₂	10 µL of specific primer mix

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6. Pipet carefully in the solution at the bottom of the tube. Use a fresh sterile pipette tip for each tube. Close all of the tubes. At this stage have another analyst witness the sample set-up.
7. **Note: Use a new sterile filter pipet tip for each sample addition. Open only one tube at a time for sample addition.** The final aqueous volume in the PCR reaction mix tubes will be 50 μ L for Profiler Plus, Cofiler and YM1, and 25.8 μ L for Powerplex 16. Transfer the DNA aliquot prepared earlier to the labeled PCR reaction mix tubes. After the addition of the DNA, cap each sample before proceeding to the next tube. **Do not vortex or mix.**
8. After all samples have been added take the rack to the amplified DNA area.

C Thermal Cycling

Turn on the ABI 9700 Thermal Cycler. (See manufacturer's instructions).

Choose the following files in order to amplify each system:

Powerplex16	YM1	Cofiler	Profiler Plus
user: casewk file: powerplex	user: casewk file: ym1	user: casewk file: profiler/cofiler	user: casewk file: profiler/cofiler

Tables 1 lists the conditions that should be included in each file. If the files are not correct, bring this to the attention of the Quality Assurance Manager and a supervisor.

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Table 1 PCR Conditions for the Perkin Elmer GeneAmp PCR System 9700

<p>9700</p> <p>Powerplex16</p> <p>user: casewk file: powerplex</p>	<p>The Powerplex file is as follows:</p> <p>Soak at 95°C for 11 minutes Soak at 96°C for 1 minute</p> <p>Ramp 100% ▶ Denature at 94°C for 30 seconds Ramp 29% ▶ Anneal at 60°C for 30 seconds Ramp 23% ▶ Extend at 70°C for 45 seconds for 10 cycles then</p> <p>Ramp 100% ▶ Denature at 94°C for 30 seconds Ramp 29% ▶ Anneal at 60°C for 30 seconds Ramp 23% ▶ Extend at 70°C for 45 seconds for 22 cycles</p> <p>30 minutes incubation at 60°C. Storage soak indefinitely at 4°C</p>
<p>9700</p> <p>Cofiler/ Profiler Plus</p> <p>user: casewk file: cofiler/profiler</p>	<p>The Cofiler/Profiler Plus file is as follows:</p> <p>Soak at 95°C for 11 minutes</p> <p>28 Cycles: ▶ Denature at 94°C for 1 minute ▶ Anneal at 59°C for 1 minute ▶ Extend at 72°C for 1 minute</p> <p>45 minute incubation at 60°C. Storage soak 90 minutes at 25°C Storage soak indefinitely at 4°C</p>

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9700 YM1 user: casewk file: ym1	The YM1 file is as follows: Soak at 95°C for 10 minutes 30 Cycles: ▶Denature at 94°C for 45 seconds ▶Anneal at 58°C for 58 seconds ▶Extend at 72°C for 1 minute 15 seconds 30 minute incubation at 60°C. Storage soak indefinitely at 4°C
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9700 Instructions

1. Place the tubes in the tray in the heat block (**do not add mineral oil**), slide the heated lid over the tubes, and fasten the lid by pulling the handle forward. Make sure you use a tray that has a 9700 label.
2. Start the run by performing the following steps:

The main menu options are RUN CREATE EDIT UTIL USER. To select an option, press the F key (F1...F5) directly under that menu option.

Verify that user is set to "casewk." If it is not, select the USER option (F5) to display the "Select User Name" screen.

Use the circular arrow pad to highlight "casewk." Select the ACCEPT option (F1).

Select the RUN option (F1).

Use the circular arrow pad to highlight the desired STR system. Select the START option (F1). The "Select Method Options" screen will appear.

Verify that the reaction volume is set to **50µL** (25µL for PowerPlex 16) and the ramp speed is set to **9600 (very important)**.

If all is correct, select the START option (F1).

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The run will start when the heated cover reaches 103°C. The screen will then display a flow chart of the run conditions. A flashing line indicates the step being performed, hold time is counted down. Cycle number is indicated at the top of the screen, counting up.

Upon completion of the amplification, remove samples and press the STOP button repeatedly until the “End of Run” screen is displayed. Select the EXIT option (F5). Wipe any condensation from the heat block with a Kimwipe and pull the lid closed to prevent dust from collecting on the heat block. Turn the instrument off.

NOTE: Return the microtube rack used to set-up the samples for PCR to the PCR Set-Up Area immediately.

Turn instruments off **ONLY** when the Main Menu is displayed, otherwise there will be a “Power Failure” message the next time the instrument is turned on. If this happens, it will prompt you to review the run history. Unless you have reason to believe that there was indeed a power failure, this is not necessary. Otherwise, press the STOP button repeatedly until the Main Menu appears.

In case of an actual power failure, the 9700 thermal cycler will automatically resume the run if the power outage did not last more than 18 hours. The history file contains the information at which stage of the cycling process the instrument stopped. Consult the Quality Assurance Unit and/or the Technical Leader on how to proceed.

After the amplification process, the samples are ready to be loaded on the fluorescent instruments. They may be stored in the appropriate refrigerator at 2-8°C for a period of up to 6 months.

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D. Amplification Troubleshooting

PROBLEM: No or only weak signal from both the positive control and the test samples

Possible Cause	Recommended Action
Mistake during the amplification set up such as not adding one of the components or not starting the thermal cycler	Prepare new samples and repeat amplification step
Thermal cycler defect or wrong program used	Check instrument, notify QA team, prepare new samples and repeat amplification step

PROBLEM: Positive control fails but sample signal level is fine

Possible Cause	Recommended Action
Mistake during the amplification set up such as not adding enough of the positive control DNA	Prepare new samples and repeat amplification step
Positive control lot degraded	Notify QA team to investigate lot number, prepare new samples and repeat amplification step

PROBLEM: Presence of unexpected or additional peaks in the positive control.

Possible Cause	Recommended Action
Contamination by other samples, contaminated reagents	Notify QA team to investigate the amplification reagents, prepare new samples and repeat amplification step
Non-specific priming	Notify QA team to check thermal cycler for correct annealing settings, prepare new samples and repeat amplification step

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PROBLEM: Strong signal from the positive controls, but no or below threshold signal from DNA test sample.

Possible Cause	Recommended Action
The amount of DNA was insufficient or the DNA is severely degraded.	<p>Amplify a larger aliquot of the DNA extract.</p> <p>Re-extract the sample using a larger area of the stain or more biological fluid to ensure sufficient high molecular DNA is present.</p> <p>Concentrate the extracted DNA using a Microcon 100 ultrafiltration device as described in the Microcon section.</p>
Test sample contains PCR inhibitor (e.g. heme compounds, certain dyes).	<p>Amplify using an additional 10 units of Taq Gold polymerase.</p> <p>Amplify a smaller aliquot of the DNA extract to dilute potential Taq Gold polymerase inhibitors.</p> <p>Re-extract the sample using a smaller area of the stain to dilute potential Taq Gold polymerase inhibitors.</p> <p>Re-extract the samples using the organic extraction procedure..</p> <p>Purify the extracted DNA using a Microcon 100 ultrafiltration device as described in the Microcon section.</p>

The decision on which of the above approaches is the most promising should be made after consultation with a supervisor.

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Samples amplified with < 150 pg are considered Low Copy Number (LCN) and are amplified for 31 cycles with Identifier reagents, whereas High Copy Number (HCN) samples contain ≥ 150 pg of DNA, and are amplified with 28 cycles with Cofiler or Profiler Plus reagents.

Due to the increased sensitivity of the Identifier™ amplification as compared to that of the Cofiler® and Profiler® Plus amplifications, samples with ≥ 100 pg may be amplified with Identifier™ reagents for 28 cycles.

The PCR strategy for LCN samples is to do three parallel amplifications which are interpreted together. However, HCN samples are interpreted individually. If duplication is necessary, it may occur concurrently or subsequently depending upon the workflow. For example, duplication of property crime case samples will occur concurrently for samples amplified with less than 250 pg of DNA only.

HCN samples are amplified separately from LCN samples.

A. Paperwork preparation

NOTE: The target DNA template amounts are as follows:

Identifier 28 cycles - 500 pg/uL

Identifier 31 cycles - 100 pg/uL

To calculate the amount of template DNA and diluent to add, the following formulas are used:

$$\text{Amt of DNA (}\mu\text{L)} = \frac{\text{Target Amount}}{(\text{sample concentration})(\text{dilution factor})}$$

$$\text{The amount of diluant to add to the reaction} = 5 \mu\text{L} - \text{amt of DNA (}\mu\text{L)}$$

RG Amp Macro HI (High Sensitivity samples)

1. Open the “RGAMP MACRO HI” excel workbook and the “RG summary sheet” excel files for samples ready to be amp’d. These files can be found in the “Property Crimes” and then “Forms” folders on the network. The “RG summary sheet” is saved as the assay name.

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2. Copy the information for samples and controls only from the “summary sheet” of the “RG summary sheet” file including the tube label (if applicable), sample name, Ct value, the calculated concentration, the target date, and the IA. Paste special as values into the corresponding columns of the “RG value” sheet of the “RG Amp macro” file. The standards and calibrators need not be copied.
3. In the column entitled “type” enter the type of amplification according to the following abbreviations:
 - a. “X” for exemplars
 - b. “V” for evidence
4. Hit Ctrl+R to run the dilution macro. A window asking “Do you want to replace the contents of the destination cell?” will appear. Click “OK”.
 - a. If the macro will not run, follow the instructions in the box and select tools, macro, security, and low. The file must be closed and reopened.
 - b. The dilution macro will separate the dilution factors from the sample name to facilitate the calculation of the neat concentration of the sample
5. Hit Ctrl+G to run the sample sorting macro.
 - a. The sort macro will filter values according to the following specifications which differ depending upon the amount of template DNA.
 - i. The macro eliminates all values that are greater than 1 pg/μL and less than 20 pg/μL for LCN amplification Identifiler™ for 31 cycles.
 - ii. All values greater than 20 pg/μL for HCN amplification with Identifiler™ for 28 cycles.
 - iii. Note, for samples with greater than 100 pg/μL and less than 124 pg/μL, the macro will indicate to add 5 μL of template DNA. (In order to avoid pipeting less than 1 μL, slightly more than 500 pg of DNA will be added to the reaction.)
 - b. The extraction negatives will be sorted independently so that they may be inspected and placed at the top of the list with the associated samples when setting up the amp sheets.
 - c. Samples will be sorted into groups for ID31 evidence amp, ID28 evidence amp, and ID28 exemplar amp. The data may then be automatically populated into the amplification and 3130 run sheets by copying and pasting samples to the appropriate column on the “samples” sheet.

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- d. Samples may be added or deleted to or from the amp sheets following the macro's execution. For example, samples with less than 4 pg/ μ L or 20 pg/amp require supervisor approval for LCN amplification, and depending upon the case, may not be amplified. Refer to the amplification guidelines and RG interpretation manual to select samples and appropriate dilutions to use for amplification calculations.
- i. To delete a sample do the following:
 1. Go to the "sort" sheet in the RG AMP MACRO HI file and locate the columns relevant to the amplification system and sample type.
 2. Select the cells relevant to the sample you would like to delete.
 3. Select edit and clear contents.
 4. Do not simply delete, always use the "clear contents" function.
 - ii. To add a sample, do the following:
 1. Copy from the "RG values revised" sheet in the "RG AMP MACRO HI" file the tube label, sample name, Ct value, the calculated concentration, the target date, and the IA.
 2. Paste special these values into the appropriate columns for the amplification system of the "samples" sheet in the "RG AMP MACRO HI" file.
 3. Alternatively, a sample may be manually added by typing the sample information into the appropriate column in the "samples" sheet.
6. Select the appropriate amplification worksheet, verify the sample information and calculations, and type the name of the amplification in cell B1 as follows:
Amonthdateyear.time for example, A011106.1000
- a. The sheet will automatically calculate the number of samples that are to be amplified. This will populate cell B2 of the worksheet.
 - b. The sheet will also calculate the amount of reagents required, and the dilution factor necessary for the samples. Verify these calculations.
7. Save the sheet in the amplification sheets folder and review.
8. Print the amplification sheet. Have the sheet reviewed by a supervisor prior to set-up.

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RG Amp Macro PC (Property Crimes Samples)

1. Open the “RGAMP MACRO PC” excel workbook and the “RG summary sheet” excel files for samples ready to be amped. The “RG summary sheet” is saved as the assay name.
2. Copy the information for samples and controls only from the “summary sheet” of the “RG summary sheet” file including the tube label (if applicable), sample name, Ct value, the calculated concentration, the target date, and the IA. Paste special as values into the corresponding columns of the “RG value” sheet of the “RG Amp macro” file. The standards and calibrators need not be copied.
3. In the column entitled “type” enter the type of amplifications according to the following abbreviations:
 - a. “X” for exemplars
 - b. “V” for evidence
4. Hit Ctrl+R to run the dilution macro. A window asking “Do you want to replace the contents of the destination cell?” will appear. Click “OK”.
 - a. If the macro will not run, follow the instructions in the box and select tools, macro, security, and low. The file must be closed and reopened.
 - b. The dilution macro will separate the dilution factors from the sample name to facilitate the calculation of the neat concentration of the sample
5. Hit Ctrl+G to run the sample sorting macro
 - a. The sort macro will filter values according to the following specifications which differ depending upon the amount of template DNA.
 - i. All values that are greater than 1 pg/μL and less than 20 pg/μL for LCN amplification
 - ii. All values greater than 20 pg/μL and less than 250 pg/uL for duplicate amp with Identifiler™ for 28 cycles
 - iii. All values greater than 250 pg/uL for single amp with Identifiler™ for 28 cycles.
 - iv. The extraction negatives will be sorted independently so that they may be inspected and placed at the top of the list with the associated samples when setting up the amp sheets.
 - v. Note, for samples with greater than 100 pg/μL and less than 124 pg/μL, the macro will indicate to add 5 μL of template DNA. (In order to avoid pipeting less than 1 μL, slightly more than 500 pg of DNA will be added to the reaction.)

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- b. The data may then be automatically populated into the amplification and 3130xl run sheets by copying and pasting samples to the appropriate column on the “samples” sheet.
- c. Samples may be added or deleted to or from the amp sheets following the macro’s execution. Refer to the amplification guidelines and RG interpretation manual to select samples and appropriate dilutions to use for amplification calculations.
 - i. To delete a sample do the following:
 1. Go to the “sort” sheet in the RG AMP MACRO PC file and locate the columns relevant to the amplification system and sample type.
 2. Select the cells relevant to the sample you would like to delete.
 3. Select edit and clear contents.
 4. Do not simply delete, always use the “clear contents” function.
 - ii. To add a sample, do the following:
 1. Copy from the “RG values revised” sheet in the “RG AMP MACRO PC” file the tube label, sample name, Ct value, the calculated concentration, the target date, and the IA.
 2. Paste special these values into the appropriate columns for the amplification system of the “samples” sheet in the “RG AMP MACRO PC” file.
 3. Alternatively, a sample may be manually added by typing the sample information into the appropriate column in the “samples” sheet.
6. Select the appropriate amplification worksheet (ID28V for initial amps and ID28Vdup for duplicate amps), verify the sample information and calculations, and type the name of the amplification in cell B1 as follows: Amonthdateyear.time for example, A011106.1000
 - a. The sheet will automatically calculate the number of samples that are to be amplified. This will populate cell B2 of the worksheet.
 - b. The sheet will also calculate the amount of reagents required, and the dilution factor necessary for the samples. Verify these calculations.
7. Save the sheet in the amplification sheets folder and review.
8. Print out the amplification sheet prior to setup. Have a supervisor review the sheet, if necessary.

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Saving Amp Sheets to the Network for Additional Samples

1. Amp sheets may be saved as independent sheets for subsequent sample additions by right-clicking the corresponding tab and selecting “move or copy”. In this window, select “(new book)” in the “to book” window and check “create a copy”. Click “OK”. Go to File – Save-As and save into the appropriate folder named with the associated amplification system followed by “waiting for amp” or “ready to amp”.
2. Samples may be manually typed into these sheets or copied and pasted special from reagent sheets or consolidated from additional amp sheets of the same type at the end of each Rotorgene run.
3. When a sheet is full the analyst may fill in the appropriate information (cells shaded blue) and save the sheet as the time and date of the amp as follows: “A041207.1700”.

TABLE 1: PCR amplification input based on Rotorgene values

RG value at 1:10 dilution pg/μL	RG value neat pg/μL	Amplification Sheet	Dilution
≥ 0.4 pg/μL	≥ 4.0* to 20 pg/μL	Amplify with ID for 31 cycles*	Neat = 1
≥ 2.0 pg/μL	≥ 20 pg/μL	Amplify with ID for 28 cycles	As appropriate

* Samples providing less than 20 pg per amplification can only be amplified with permission of a supervisor.

B. Personal Preparation

1. Put on a mask, lab coat, hair covering, and gloves.
 - a. Ensure that the cuffs of the lab coat completely cover the arms.
 - b. Lab coats may be reused for up to one week.
 - c. Masks, hair coverings and gloves may be disposed after one use.
2. **Do not enter room without appropriate attire. Never touch any apparatus, bench etc with bare hands.**
3. Do not hesitate to change gloves or at least rinse gloves with bleach followed by ETOH if contamination is suspected or after using the phone in the lab or touching door handles.

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C. Work Place Preparation

1. If using a plate, turn on the heated plate seal apparatus.
2. Apply 10% bleach followed by water, and 70% ETOH to the entire work surface and pipets. All amp setup steps should be carried out under a dead air or ventilated hood.
3. Retrieve clean racks and cap openers, and irradiated microcentrifuge tubes, PCR tubes and/or plates and irradiated water.
4. If using a PCR plate, place an easy pierce strong seal on top of the plate, and place the plate in the plate adapter on the ABgene heat sealer.
 - a. Push the heat sealer on top of the plate for 2 seconds.
 - b. Rotate the plate and reseal for 2 additional seconds.
5. **Arrange work place to minimize crossover.**
6. Position gloves nearby with bleach/ETOH/water in order to facilitate frequent glove changes and cleaning.

D. Sample Preparation

1. Prepare dilutions for each sample if necessary according to table 2.

TABLE 2: Dilutions

Dilution	Amount of DNA Template (uL)	Amount of Irradiated Water (uL)
0.1	2	18
0.2	2	8
0.05	2.5	47.5
0.01	2	198

- a. Centrifuge samples at full speed briefly.
- b. Label tubes appropriately for dilutions. Add the correct amount of irradiated water as specified by the amplification sheet.
- c. Pipet sample up and down several times to thoroughly mix sample. Avoid vortexing sample and the possible creation of aerosols.
- d. Set the sample aside until you are ready to aliquot it for amplification.

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2. **For an Identifiler™ 28 cycle amplification, make a 0.5 (1/2) dilution of the ABI Positive (A9947) control at 100 pg/ µL (5 µL in 5 µL of water).** This yields 50 pg/µL of which 5 µL or 250 pg will be used.
 - a. If only half a plate of samples are amplified, only one PE is necessary, however to encompass all of the injections required for a full plate of samples, amplify two PEs (10 µL in 10µL of water).
 - b. The amp sheet will automatically populate these PEs.
3. **For an Identifiler™ 31 cycle amplification, make a 0.2 (1/5) dilution of the ABI Positive (A9947) control at 100 pg/µL (2 µL in 8µL of water).** This yields 20 pg/µL of which 5 µL or 100 pg will be used.
4. Irradiated water will serve as an amplification negative control.
5. Arrange samples in a 96 well rack in precisely the positions they appear on the sheet.
6. **Witness step.** Ensure that your samples are properly positioned.

E. Master Mix Preparation

1. Retrieve **Identifiler™** primers and reaction mix from the refrigerator, and Taq Gold from freezer, and store in a Nalgene cooler on bench. **Record the lot numbers of the reagents.**
2. Vortex or pipet the reagents up and down several times to thoroughly mix the reagents. After vortexing, centrifuge reagents at full speed briefly to ensure that no sample is trapped in the cap. Do not vortex TAQ GOLD as it may degrade the enzyme.
3. Consult the amplification sheet for the exact amount of Identifiler™ Primers, Reaction mix, and TAQ GOLD, to add. The amount of reagents for one amplification reaction is listed in Table 3.

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TABLE 3: Identifiler™ PCR amplification reagents for one sample

Reagent	Per reaction
Primer mix	2.5µL
Reaction mix	5uL
AmpliTaq Gold DNA Polymerase (5U/µL)	0.5uL
Mastermix total:	8uL
DNA	5 µL

F. Reagent and Sample Aliquot

1. Vortex master mix to thoroughly mix. After vortexing, tap or briefly centrifuge master mix tube on bench to ensure that no reagent is trapped in the cap.
2. Add **8 µL** of the Identifiler™ master mix to each tube or well of the 96 well plate that will be utilized, changing pipette tips and remixing master mix as needed.

NOTE: Use a new sterile filter pipet tip for each sample addition. Open only one tube at a time for sample addition.

3. Prior to immediately adding each sample or control, pipet each sample or control up and down several times to thoroughly mix. The final aqueous volume in the PCR reaction mix tubes will be 13µL. After addition of the DNA, cap each sample before proceeding to the next tube.
4. Sealing the plate
 - a. If using a PCR plate, place a super pierce strong seal on top of the plate, and place the plate in the plate adapter on the ABgene heat sealer.
 - b. Push the heat sealer on top of the plate for 2 seconds.
 - c. Rotate the plate and reseal for 2 additional seconds.
 - d. Label the plate with the A for amplification and the date and time.
(A011104.1300)
5. After all samples have been added, return DNA extracts to storage and take the rack or plate to the amplified DNA area.

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G. Thermal Cycling

1. Turn on the ABI 9700 Thermal Cycler. (See manufacturer's instructions).
2. Choose the following files in order to amplify each system:

Identifiler 28	Identifiler 31
user: hisens or casewk file: id28	user: hisens or casewk file: id31

3. Tables 4 lists the conditions that should be included in each file. If the files are not correct, bring this to the attention of the Quality Assurance Manager and a supervisor.

Table 4 PCR Conditions for the Perkin Elmer GeneAmp PCR System 9700

9700	The Identifiler file is as follows:
Identifiler 28 or 31	Soak at 95°C for 11 minutes
user: hisens or casewk file: id28 or id31	28 or 31 Cycles: ▶Denature at 94°C for 1 minute ▶Anneal at 59°C for 2 minutes ▶Extend at 72°C for 1 minute
	60 minute incubation at 60°C.
	Storage soak indefinitely at 4°C

9700 Instructions

1. Place the tubes in the tray in the heat block (**do not add mineral oil**), slide the heated lid over the tubes, and fasten the lid by pulling the handle forward. Make sure you use a tray that has a 9700 label.

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2. Start the run by performing the following steps:
 - a. The main menu options are RUN CREATE EDIT UTIL USER. To select an option, press the F key (F1...F5) directly under that menu option.
 - b. Verify that user is set to "hisens" or "casewk." If it is not, select the USER option (F5) to display the "Select User Name" screen.
 - c. Use the circular arrow pad to highlight "hisens" or "casewk." Select the ACCEPT option (F1).
 - d. Select the RUN option (F1).
 - e. Use the circular arrow pad to highlight the desired STR system. Select the START option (F1). The "Select Method Options" screen will appear.
 - f. Verify that the reaction volume is set to **13µL** and the ramp speed is set to **9600 (very important)**.
 - g. If all is correct, select the START option (F1).

The run will start when the heated cover reaches 103°C. The screen will then display a flow chart of the run conditions. A flashing line indicates the step being performed, hold time is counted down. Cycle number is indicated at the top of the screen, counting up.

3. Record thermalcycler number on the worksheet and fill in the thermalcycler logbook.

NOTE: Return the microtube rack used to set-up the samples for PCR to the PCR Set-Up Area immediately.

Upon completion of the amplification, remove samples and press the STOP button repeatedly until the "End of Run" screen is displayed. Select the EXIT option (F5). Wipe any condensation from the heat block with a Kimwipe and pull the lid closed to prevent dust from collecting on the heat block. Turn the instrument off.

Turn instruments off **ONLY** when the Main Menu is displayed, otherwise there will be "Power Failure" message the next time the instrument is turned on. If this happens, it will prompt you to review the run history. Unless you have reason to believe that there was indeed a power failure, this is not necessary. Otherwise, press the STOP button repeatedly until the Main Menu appears.

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In case of an actual power failure, the 9700 thermal cycler will automatically resume the run if the power outage did not last more than 18 hours. The history file contains the information at which stage of the cycling process the instrument stopped. Consult the Quality Assurance Unit and/or the Technical Leader on how to proceed.

After the amplification process, the samples are ready to be loaded on the fluorescent instruments. They may be stored in the appropriate refrigerator at 2-8°C.

H. Clean up

1. Wipe entire work area surface and pipets with bleach followed by water and ETOH.
2. Clean or soak any racks or cap openers with bleach and ETOH.
3. If necessary, replenish the microcentrifuge, PCR, or water tube supply by irradiating the empty tubes and water tubes for 30, 10, or 45 minutes, respectively. Consult the irradiation protocol for further details.
4. Turn on the UV lamp in the hood for ten minutes.

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I. Amplification Troubleshooting

PROBLEM: No or only weak signal from both the positive control and the test samples

Possible Cause	Recommended Action
Mistake during the amplification set up such as not adding one of the components or not starting the thermal cycler	Prepare new samples and repeat amplification step
Thermal cycler defect or wrong program used	Check instrument, notify QA team, prepare new samples and repeat amplification step

PROBLEM: Positive control fails but sample signal level is fine

Possible Cause	Recommended Action
Mistake during the amplification set up such as not adding enough of the positive control DNA	Prepare new samples and repeat amplification step
Positive control lot degraded	Notify QA team to investigate lot number, prepare new samples and repeat amplification step

PROBLEM: Presence of unexpected or additional peaks in the positive control.

Possible Cause	Recommended Action
Contamination by other samples, contaminated reagents	Notify QA team to investigate the amplification reagents, prepare new samples and repeat amplification step
Non-specific priming	Notify QA team to check thermal cycler for correct annealing settings, prepare new samples and repeat amplification step

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PROBLEM: Strong signal from the positive controls, but no or below threshold signal from DNA test sample.

Possible Cause	Recommended Action
The amount of DNA was insufficient or the DNA is severely degraded.	<p>Amplify a larger aliquot of the DNA extract.</p> <p>Re-extract the sample using a larger area of the stain or more biological fluid to ensure sufficient high molecular DNA is present.</p> <p>Concentrate the extracted DNA using a Microcon 100 ultrafiltration device as described in the Microcon section.</p>
Test sample contains PCR inhibitor (e.g. heme compounds, certain dyes).	<p>Amplify using an additional 10 units of Taq Gold polymerase.</p> <p>Amplify a smaller aliquot of the DNA extract to dilute potential Taq Gold polymerase inhibitors.</p> <p>Re-extract the sample using a smaller area of the stain to dilute potential Taq Gold polymerase inhibitors.</p> <p>Re-extract the samples using the organic extraction procedure..</p> <p>Purify the extracted DNA using a Microcon 100 ultrafiltration device as described in the Microcon section.</p>

The decision on which of the above approaches is the most promising should be made after consultation with a supervisor.

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5. GENERAL GUIDELINES FOR FLUORESCENT STR ANALYSIS

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Batch processing

1. Exemplars and evidence samples must be handled separately at all times. These samples must never be together on the same sample tray.
2. For the ABI 3100, it is acceptable to load one exemplar and one evidence tray in the same instrument. Two trays are the equivalent of two consecutive runs.
3. Samples from one amplification sheet should be processed together, so that each sample is accompanied by the appropriate controls.
4. Use the correct worksheet for the specific sample type, and make sure the sample preparation set-up is witnessed properly.
5. Samples that need to be rerun must be loaded on a run with the appropriate sample type. It is not necessary to rerun all of the amplification controls for the repeated sample.

ATTENTION: Each capillary run that is performed must have at least one correct positive control.

Sample handling

1. Prior to loading on the capillary, the amplified samples are stored at +4°C in the amplified DNA area. The tubes containing the amplified product must never leave the amplified DNA area.
2. Amplified samples that have been loaded on an instrument should be stored until the electrophoresis results are known. After it has been determined that the amplified samples do not require repeated testing, they may be discarded.

Instrument and computer maintenance

1. Be gentle with all instrument parts and instruments. Keep everything clean.
2. It is good practice to monitor initial instrument performance. This enables the user to detect problems such as leaks, air bubbles or calibration issues.

FORENSIC BIOLOGY PROTOCOLS FOR FORENSIC STR TYPING

5. GENERAL GUIDELINES FOR FLUORESCENT STR ANALYSIS

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3. The Macintosh computer should be restarted before each run. This prevents the hard drive from becoming too fragmented.
4. PCs need to be maintained through regular defragmentation.
5. Delete data files and other non-essential files from the hard disk at least once a week.
6. Notify the Quality Assurance Unit if any problems are noted.

ARCHIVED

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6. STR DATA ARCHIVING		
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STR DATA ARCHIVING ON JAZ DISKS FOR MACINTOSH BASED SYSTEMS (ABI 310)

In absence of a MAC network, JAZ disks are used as transport medium from the data collection instruments to the external analysis stations. A different set of JAZ disks are used as a final archive after analysis has been completed. Always transfer the run files onto the computer hard drive before processing; otherwise the JAZ disks will get corrupted.

Data transfer

1. Before copying a run, identify the **Run folder** containing the project file based on the date and time noted on the gel or sample sheet. At this point the folder can be renamed with the gel/sample sheet name, which will make it easier to find later.
2. Put a designated JAZ archiving disk in the JAZ drive. The disk icon will be visible on the desktop. Drag the icon of the renamed run folder onto the JAZ disk icon. After copying is complete, drag the JAZ disk icon onto the trash icon to expel the disk.

NOTE: Always remove external disks by dragging the disk icon into the trash icon. Do not press the expel button on the JAZ disk drive!!!! Do not use paperclips!!!!

Restarting the computer can help if the JAZ disk icon will not appear on the desktop.

3. Copy the run on external analysis station hard drive and delete it from the transfer disk.

Data archiving

1. After the gel or project has been analyzed and processed with Genotyper, the folder should contain the following items: An injection list, run log, a project file, sample files for all samples, and the Genotyper files

Make sure the folder is complete.

2. The folder should have been renamed (see Genotyper instructions), according to the following format:

Instrument name Files (e.g. CE1/04-002 files)

3. Save folder on instrument specific archiving disk or Y STR disk.

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4. After the copy process is complete, delete the run folders from the collection instrument and the external analysis station computer hard drive.
5. JAZ disks are rewritable and the Genotyper files can be opened, reedited and saved. Reediting is documented on the hard copies.

STR DATA ARCHIVING ON CD'S FOR WINDOWS BASED SYSTEMS (ABI 3100)

The external analysis stations for 3100 data are networked together but CD's need to be burned to transfer data from the collection instruments to the analysis stations.

A. Formatting of transfer CD

When a blank CD-R or CD-RW disc is formatted by **DirectCD**, it may be used in the same way as any other removable media such as JAZ, ZIP, or floppy discs. A DirectCD formatted CD-R/CD-RW can have data added and deleted provided it is in a CD-RW drive. You may work and read off the disc in both CD-ROM and CD-RW drives.

To format a CD-R or CD-RW in DirectCD:

1. Insert a blank CD-R or CD-RW disc into the CD-RW drive.
2. Wait for the program **Easy CD Creator 4** to launch itself.*
3. Click **DATA**.
4. Click **DIRECTCD**. The **Adaptec DirectCD Wizard** will open.

*If Easy CD Creator 4 does not open within 30 seconds, the Adaptec DirectCD Wizard may be opened directly from the **START** menu in the **Programs** category under the heading **DirectCD**.

5. Click **Next>**. The **Drive Information** window will open.
6. Click **Next>**. The **Format Disc** window will open.
7. Click **Next>**. The **Name Your Disc** window will open. Give the disc a name up to eleven characters long.

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6. STR DATA ARCHIVING		
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8. Click **Finish**. You will receive the message, “Disc in Drive _: has been successfully formatted.”
9. Click **OK**. Your disc is now ready to write on and delete from (only in CD-RW drives), as well as read from and worked off of (in both CD-RW and CD-ROM drives). It behaves in the same way as the floppy discs, JAZ/ZIP.
10. You can save onto it from a program’s file menu or you can add/delete files to/from the disc through **My Computer, Windows Explorer**, etc...
11. When a DirectCD disc is in the CD-RW drive, a small red spot appears on the DirectCD icon (a CD on top of a disc drive) in the bottom right-hand side of the screen.

To transfer casework runs, there are shortcuts on the desktops for faster access to the CD-RW drives. You can ‘Drag-and-drop’ files directly from the Current Runs folder onto the shortcut icons to copy them onto the disc. Double-click the shortcut icon to view the data on the disc.

B. Copy and transfer of files

1. When a run is finished, locate the injection folders on the 3100 instrumental computer that were generated during the course of the run. These folders are in the Current Runs folder that is located on the desktop of the instrumental computer.
2. Create and organize a new folder for each amplification system used (e.g., cofil, profiler plus) to contain the injection folders. Name this folder(s) with the filename according to your sample sheet (e.g., Stripes04-009Co). This new folder will be referred to as the **common runs folder(s)**.

Keep in mind that when two or more different amplification systems are used, several common runs folders will be needed (see examples below).

Example #1: A Cofil and a Profiler Plus amplification set were run on Stars with the following sample sheet name: Stars04-008Co-003Pro. Two common run folders will need to be created with the following names: Stars04-008Co and Stars 04-003Pro.

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6. STR DATA ARCHIVING		
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Example #2: Two Cofiler amplification sets were run on Stars with the following sample sheet name: Stars04-010Co. Only one common run folder needs to be created with the same name (Stars04-010Co) even though following analysis, two genotyper files will be generated with names corresponding to each amplification set as follows: Stars04-010CoA and Stars04-010CoB.

Note: Be sure to also include a copy of the run's sample sheet in the common runs folder. Sample sheets are located in the sample sheet folder on the desktop. It is necessary to save only the .xls file (**not** the .plt file). If more than one common runs folder is created, then each should contain a copy of the sample sheet.

3. Insert a CD-RW transfer disk into the CD-RW drive of the instrumental computer.
4. Copy the common runs folder(s) onto the CD-RW disk by clicking and dragging the common runs folder(s) onto the CD-RW drive icon located on the desktop.
5. Eject the CD-RW transfer disk from the instrumental computer and take it over to the analysis network for further analysis.
6. Insert the CD-RW transfer disk into the CD-R or CD-RW drive on analysis station #1, #2, or #3.
7. View the desired common runs folder(s) by double-clicking the CD icon that is located on the desktop. Copy the desired common runs folder(s) to the PC hard drive by clicking and dragging the common runs folder(s) from the transfer disk to the **Work folder** (located on the desktop).

Note: This work folder is specific for each terminal and cannot be accessed from the other two terminals.

8. Proceed with the project and genotyper analysis according to the protocol described in the **STR Analysis on the 3100 Capillary Electrophoresis Genetic Analyzer** and **Genotyper** sections of the STR manual.

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6. STR DATA ARCHIVING		
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9. After analysis is completed and Genotyper files have been saved in the Common run folder, copy the Common run folder to the **Archive folder**. These folders are organized on each analysis station according to the following hierarchy:

- **Archive folder**
- Instrument folders (e.g., Stars, Stripes; located in the **Archive folder**)
- Amplification system folders (e.g. Cofiler, Profiler Plus; located in the instrument folders)

The **Archive folder** is physically present on analysis station #1 but is shared with analysis stations #2 and #3 on the analysis network. This means that even though the data is being copied from the **Work folder** to the **Archive folder** at analysis station #2 or #3, it is physically being copied to the hard drive of analysis station #1. Along the same lines, editing data (see Section E.) can be performed at any of the analysis workstations.

10. Delete completed folders from Work folder and Transfer disk. Return the transfer disk back to the 3100 instrument area.

C. Temporary Archive (weekly)

At the end of each week it is the responsibility of the STR station supervisor(s) to do the following:

1. Copy all of the analyzed common runs folders present in the **Archive folder** to a backup folder present on analysis station #2. This folder (located in the D drive of analysis station #2) will not be accessed during routine run analysis and is maintained only for the purpose of having a backup copy. This backup procedure can only be done on analysis station #2.
2. Delete runs that have already been analyzed and backed-up on analysis station #2 from the 3100 instrumental computers

D. Permanent Archive (every three months)

At the end of each 6-month span (e.g. Jan-Jun, Jul-Dec) with a lag time of 3 months (e.g., Oct, Apr) it is the responsibility of the Forensic Biology computer support analyst(s) to do the following:

1. Two CD-Rs will each be burned to contain all of the analyzed common runs folders present on analysis station #1 for the given 6 month period. One set of copies will be stored at 520, the other off-site at Bellevue.

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6. STR DATA ARCHIVING		
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IMPORTANT: Never fill the CD-R disks with more than 675 Mb of data. This will insure that with the inclusion of future edits, all of the data will still fit on one CD-R disk during the reburning process.

2. After all of the analyzed common runs folders for the 6 month period have been burned and verified, copies of the burned files will be deleted the analysis network.

Note: *The end result in the storage of files is that at any given time there will be 2 copies of each run saved either on 2 separate hard drives or on 2 separate CD-R disks.*

E. Editing stored runs

If an edit(s) needs to be done on a previously analyzed run whose copy is still present in the Archive folder of analysis station #1, do the following:

1. Make all necessary edits to the relevant file(s) present in the **Archive folder** on the desktop of the analysis station.

Note: Due to file sharing on the analysis network, editing can be done from any of the analysis stations on the analysis network.

2. Any changes made in item 1 will be backed-up onto analysis station #2 according to the weekly backup procedure described above.

If an edit(s) needs to be done on a previously analyzed run that has already been burned on CD-R, do the following:

1. Find the desired CD-R disk at the analysis network area and copy all of its contents to the Temp Archive folder located on the desktop of the analysis workstation. This will be used as a temporary working file. Make edits to the desired file(s) present in the temporary working file.
2. After all of the necessary edits are done, re-burn two new CD-R disks to contain all of the contents of the temporary working file including the changes that were made. One of the CD-Rs will replace the one at the analysis network area and the other will replace the backup at Bellevue. After the original disks are replaced with the two new disks, the original disks should be destroyed (e.g. break into two or feed into an appropriate shredder) and discarded. After new copies of disks have been burned, delete all of the files from the Temp Archive folder on the desktop.

FORENSIC BIOLOGY PROTOCOLS FOR FORENSIC STR TYPING

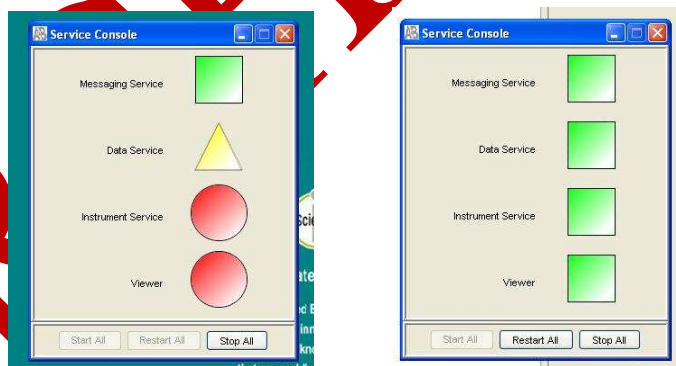
7. STR ANALYSIS ON THE ABI 3130xl GENETIC ANALYZER

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I. Setting Up A 3130xl Casework Run

1. Collect amp sheets that are ready to run and/or check rerun sheets for samples that need to be rerun. Evidence and exemplars must be run on separate plates.
2. Turn on or restart the computer attached to the instrument.
3. Press “CTRL-ALT-DEL” to login.
4. User should be “Administrator”, password should be left blank.
5. Press “OK”.
6. Open the 3130xl Data Collection v3.0 software by double clicking on the desktop Icon or select **Start > All Programs > AppliedBiosystems > Data Collection > Run 3130xl Data Collection v3.0** to display the **Service Console**.

By default all applications are off indicated by the red circles. As each application activates, the red circles (off) change to yellow triangles (activating), eventually progressing to green squares (on) when they are fully functional.



NOTE: This process could take several minutes. The **Service Console** must not be closed or it will shut down the application.

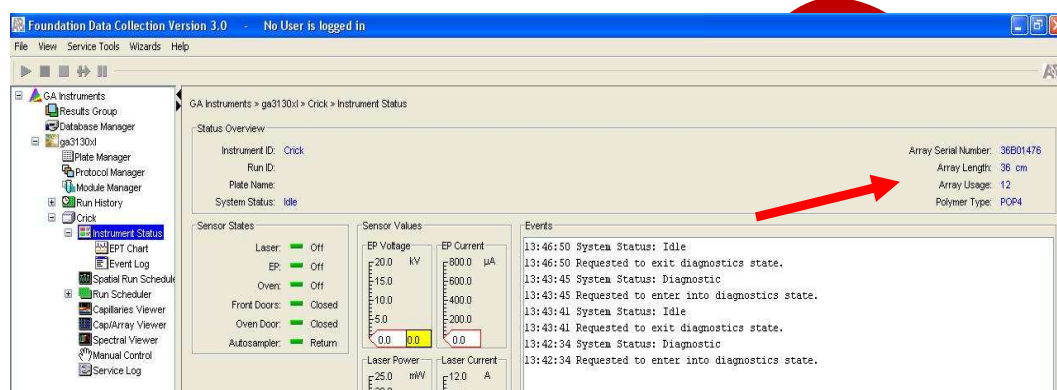
Once all applications are running, the **Foundation Data Collection** window will be displayed at which time the **Service Console** window may be minimized.

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7. STR ANALYSIS ON THE ABI 3130xl GENETIC ANALYZER

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7. Check the number of injections on the capillary in the 3130XL binder and in the **Foundation Data Collection** window by clicking on the **ga3130xl > instrument name > Instrument Status**. If the numbers are not the same, update the binder. If the number is ≥ 140 , notify QC. Proceed only if the number of injections you are running plus the usage number is ≤ 150 .

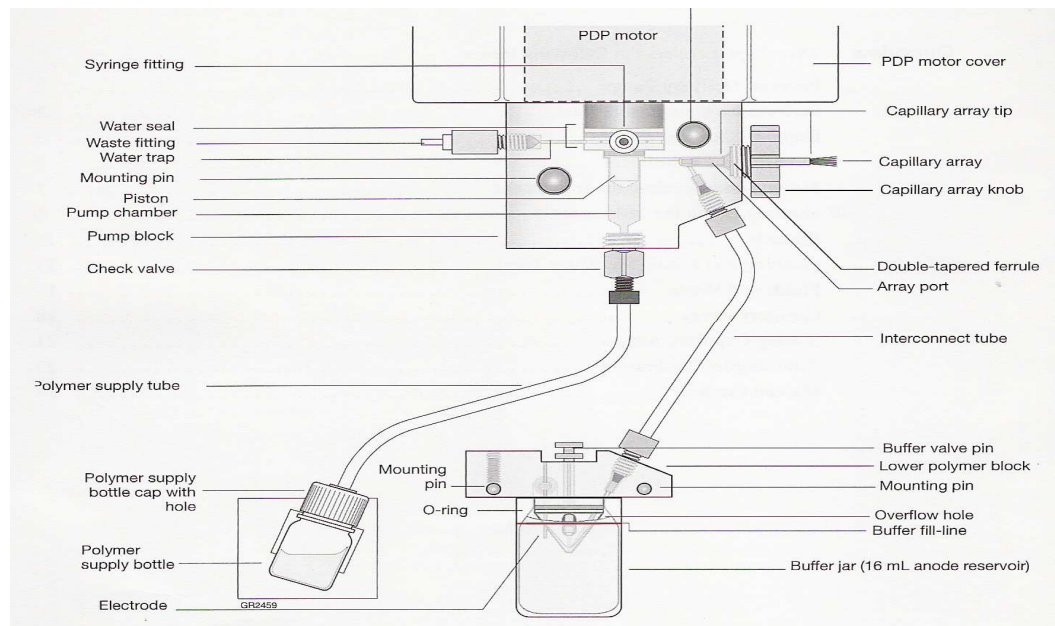


8. Check the binder to see when the POP4 was last changed. If it is >7 days, proceed with POP4 change (See Part XIII of this Section) and then return to Step 11.
9. Check the level of POP4 in the bottle to ensure there is enough for your run (~ 450 μL for 6 injections). A full piston chamber is approximately 200ul. If there is not, proceed with POP4 change (See Part XIII of this Section) and then return to Step 10.

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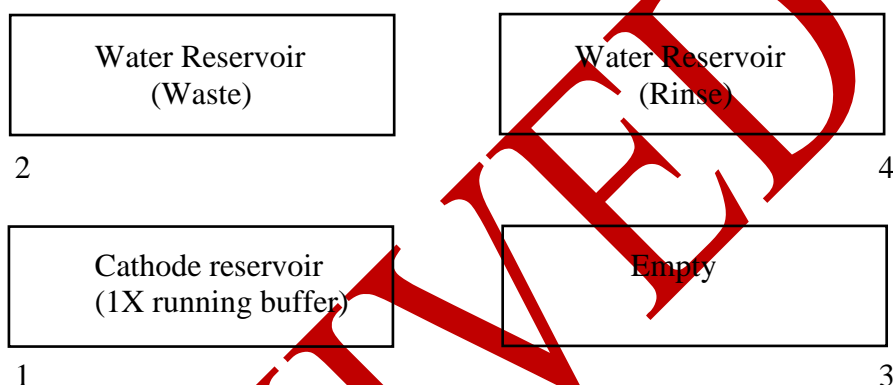


10. If you are the first run on the instrument of the day, proceed with step 11-20. If a run has already been performed on the instrument that day and the "buffer changed" column has been checked off in the binder, skip to **"Creating a Plate Record through Excel"**.
11. Close the instrument doors and press the tray button on the outside of the instrument to bring the autosampler to the forward position.
12. Wait until the autosampler has stopped moving and then open the instrument doors.
13. Remove the three plastic reservoirs in front of the sample tray and anode jar from the base of the lower pump block and dispose of the fluids.
14. Rinse and fill the "water" and "waste" reservoirs to the line with Gibco® deionized water.
15. Make a batch of 1X buffer (45 ml Gibco® water, 5 ml 10X buffer) in a 50 mL conical tube. Record the lot number of the buffer, date of make, and initials on the side of the tube. Rinse and fill the "buffer" reservoir and anode jar with 1X running buffer to the lines.

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16. Dry the outside and inside rim of the reservoirs/septa and outside of the anode jar using a Kimwipe and replace the septa strip snugly onto each reservoir. **If these items are not dry arcing could occur thus ruining the capillary and polymer blocks.**
17. Place the reservoirs in the instrument in their respective positions, as shown below:



18. Place the anode jar at the base of the lower pump block.
19. Close the instrument doors.
20. In the binder, check the “buffer changed” column.

II. Creating a Plate Record through Excel

This excel sheet will be imported into the 3130XL Data Collection v3.0 software.

1. Double click on the **Sample Sheet** icon on the desktop to open the sample sheet template. Make sure that the “Sample Sheet” tab is selected (bottom left of the screen). This is a ‘Read Only’ file so changes can not be saved under this file name.

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NOTE: The Sample Sheet has the following defaults to ensure that all fields contain entries for a successful import into the Data Collection software.

Case Number- Sample Description	“BLANK”
Tube Label	“E”
Sys.	“C”
Type	“S”
IA	“E”

Microsoft Excel - Crick Sample Sheet [Read-Only]

File Edit View Insert Format Tools Data Window Help

3130XL Crick Capillary Electrophoresis Run Control Sheet

Sample Sheet Name: No Spaces in "Sample Sheet Name", Use Only -_0{}#.+

Case Number-Sample Description

Tube Label

Sys.

type

IA

Lot Numbers:

POP4

Buffer

Formamide

The Excel sheet must be completely filled in order for the plate record to be imported to the 3130xl Data Collection v3.0 Software so the “Blanks” must remain present in all cells unless sample information is present.

- Fill in the sample sheet name in cell E3. Casework sample sheets should be named indicating the instrument, the year and the consecutive run number for the multiplex, e.g.

Crick06-021Co-009Pro or Watson06-004Co or Crick06-015Pro

- Fill in the sample information in the “Case Number-Sample Description” and “Tube Label” columns. Room has already been reserved for the Allelic Ladders in wells A1, A3, A5, A7, A9, A11 (the grey cells) and the positive control in well B1 (the positive control may be edited, though the ladders may not, as it is mandatory that there be a ladder included with each injection set for Cofiler and Profiler).

IMPORTANT: When naming in the “Case Number-Sample Description” and “Tube Label” columns, you may use letters, numbers, and only the following characters: - _ . () { } [] + ^ . Do not use commas, colons, or quotes. Use the character ^ instead of quotes.

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Also, **NO SPACES** between characters are allowed. If an incorrect character is used in the sample sheet name, you will not be allowed to import the plate record into the 3130XL Data Collection v3.0 software.

Ex. FB06-XXXX_Tshirt_st._1A1 or FB06-XXXX_Vag_EC

4. In the “Sys.” column, fill in the appropriate letter for the system in which the sample was amplified:

Amp System	Designation
Cofiler	C
Profiler Plus	P
YM1	M
Identifiler 28	I
Cofiler Rerun	CR
Profiler Plus Rerun	PR
YM1 Rerun	MR
Identifiler 28 Rerun	IR

5. In the “type” column, fill in the appropriate letter/s for the type of the sample:

Sample Type	Designation
Allelic Ladder	AL
Positive Control	PC
Negative Control	NC
Sample	S

6. Proofread your sample sheet, make corrections and re-save as necessary.
7. Save the sample sheet by selecting **Save As** from the File menu in the format of: *yoursamplesheetname.xls*. Save in D:\AppliedBiosystems\Sample Sheets (.xls files.)

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8. Click on the **Second Tab** labeled “test” on the bottom left of the screen. Select **Save As** from the File menu and do the following:
 - Change **Save as file type** to “Text (Tab-delimited)”.
 - Save in D:\AppliedBiosystems\Plate Records (.plt files.)
9. Click **OK** (The selected file type does not support workbooks that contain multiple sheets)
10. Click **Yes** (Do you want to keep the workbook in this format?)
11. To quit Excel, **File > Exit**. You will be prompted to save again by Excel; this is not necessary select **NO**.

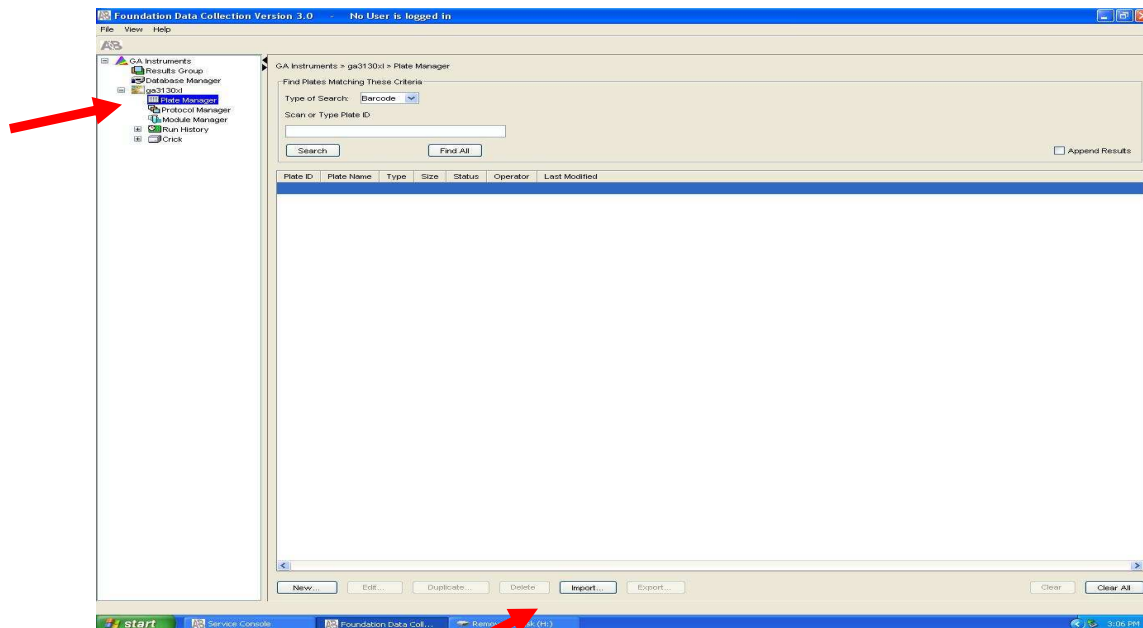
III. Foundation Data Collection (Importing Plate Record)

1. Maximize the Foundation Data Collection window.
2. Click + to expand subfolders in the left tree pane of “ga 3130xl”. All application folders are now visible.
3. Click on “**Plate Manager**”.
4. In the Plate Manager window click on “**Import...**”

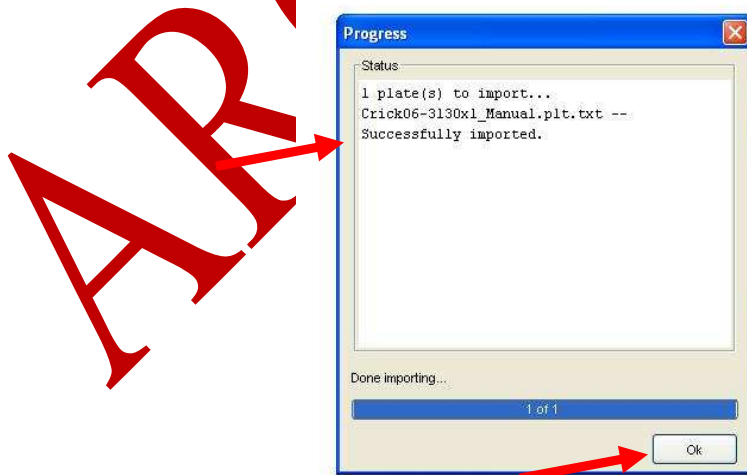
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5. Browse for your plate record in D:\AppliedBiosystems\Current Runs. Double click on your file or highlight it and click **Open**.
6. If you followed all the rules and correctly created your Plate Record a window will prompt you that you have successfully imported your plate record. Click **OK**.



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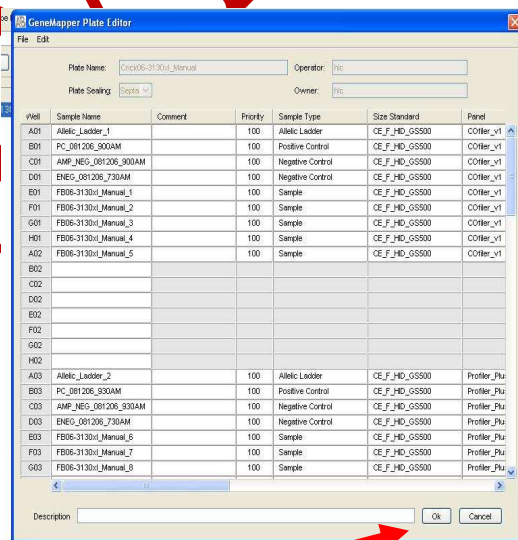
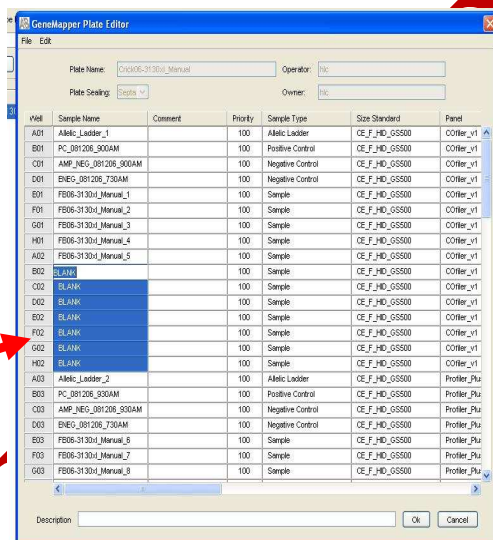
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If the Plate Record will not import, a window will prompt you where you need to make your changes. Go back to edit your Excel sheet and resave your Plate Record with the same file name.

NOTE: Due to a program issue when importing a plate record to the 3130xl Data Collection v3.0 software, in order for the instrument not to run “BLANK” as a regular sample, you must follow steps 7-12.

- Click on “**Edit...**” or double click on your file to go to the Edit window.
- Highlight the “**BLANK**”(s) in the Sample Name column and hit the **Shift and Delete** button to clear the contents. When all the “**BLANK**” columns are cleared, hit **OK** on the bottom of the window.



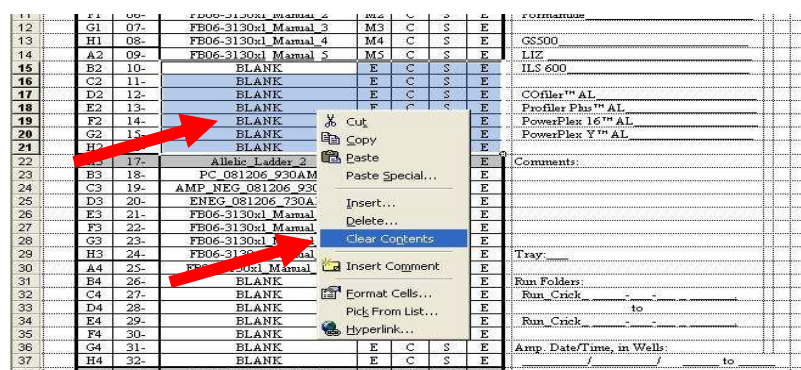
- Restore Down or Minimize the Foundation Data Collection window.
- Open the Sample Sheet folder shortcut on the desktop to edit your Sample sheet.

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- Highlight all the labeled “**BLANK**” columns and all the columns corresponding to those samples. Right click on the highlighted area and click on “**Clear Contents**”. Do not save this change.



- Print your Excel Sample Sheet.
- Close Microsoft Excel.

IV. Preparing and Running the DNA Samples

- Retrieve amplified samples from the thermalcycler or refrigerator. If needed, retrieve a positive control from a previous passing run for reruns.
- If condensation is seen in the caps of the tubes, centrifuge tubes briefly.
- Arrange amplified samples in a 96-well rack according to how they will be loaded into the 96-well reaction plate. Sample order is as follows: A1, B1, C1, D1... G1, H1, A2, B2, C2...G2, H2, A3, B3, C3, etc. Thus the plate is loaded in a columnar manner where the first injection corresponds to wells A1-H2, the second A3-H4 and so on.
- If rerunning high, prepare 1/10 dilutions of allelic ladder and positive control.
- Label the side of the reaction plate with the name used for the Plate Record with a sharpie and place the plate in the plate base.

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6. Refer to the tables below to determine the amount of HiDi Formamide and appropriate standard to use for the number of samples you have (tables list reagent amount for n+2 samples; thus extra will remain in the tube). To prepare mix for (n+2) samples, 9.5 μ L of HiDi Formamide + 0.5 μ L of GS500 Rox Standard or Liz Standard is mixed per sample. **HiDi Formamide cannot be refrozen.**

Cofiler & Profiler

# Samples + 2	HiDi Form	GS500 Std
16	171 μ L	9 μ L
32	323 μ L	17 μ L
48	475 μ L	25 μ L
64	627 μ L	33 μ L
80	779 μ L	41 μ L
96	931 μ L	49 μ L
112	1083 μ L	57 μ L
128	1235 μ L	65 μ L

YM1

# Samples + 2	HiDi Form	LIZ Std
16	171 μ L	9 μ L
32	323 μ L	17 μ L
48	475 μ L	25 μ L
64	627 μ L	33 μ L
80	779 μ L	41 μ L
96	931 μ L	49 μ L
112	1083 μ L	57 μ L
128	1235 μ L	65 μ L

7. Aliquot 10 μ L of the formamide/standard mixture into each well being used on the 96-well reaction plate.
8. If an injection has less than 16 samples, add 12ul of either dH₂O, buffer or formamide/standard mix to all unused wells within that injection.
9. Rerun "high" samples **cannot** be on the same injection as non rerun samples. Rerun "normal" samples may be integrated with non rerun samples.
10. Have someone witness the tube setup by comparing the tube labels and positions indicated on the sample sheet with the tube labels and positions of the tubes themselves.

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11. Add samples to the plate, adhering to the following guidelines:

For samples being run at normal parameters:

- a. add 1 µL of allelic ladder
- b. add 2 µL of PCR product (including the positive control)

For samples being rerun normal at a 1/10 dilution:

- a. add 2 µL of a 1/10 dilution

For samples being run at rerun high parameters:

- a. add 4 µL of a 1/10 dilution of the positive control and/or allelic ladder
- b. add 4 µL of PCR product

12. When adding PCR product, make sure to pipette the solution directly into the formamide and gently flush the pipette tip up and down a few times to mix it.
13. Once all of the samples have been added to the plate, place a new 96-well Septa over the reaction plate and firmly press the septa into place.
14. Spin plate in centrifuge at 1000 RPM for one minute.
15. Place the plate on a 9700 thermocycler. (Make sure to keep the thermocycler lid off of the sample tray to prevent the septa from heating up.)
16. Select the “denature/chill” program.
17. Make sure the volume is set to 12 µL.
18. Press **Run** on the thermocycler. The program will heat denature samples at 95°C for 5 minutes followed by a quick chill at 4°C (this will run indefinitely, but the plate should be left on the block for at least **5 min**).
19. While the denature/chill is occurring, you can turn on the oven.

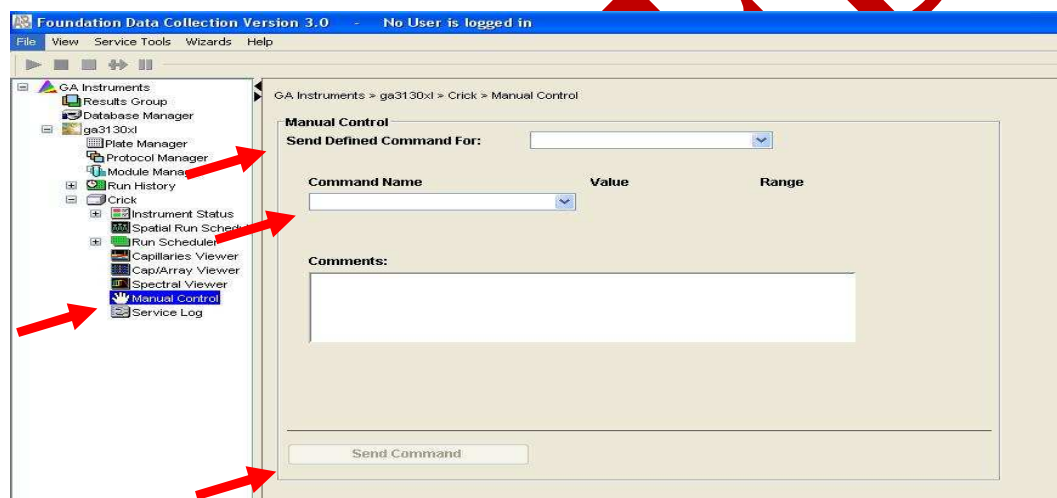
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V. Turning the Oven on and Setting the Temperature

1. In the tree pane of the Data Collection v3.0 software click on **GA Instrument > ga3130xl > instrument name > Manual Control**
2. Under Manual Control “**Send Defined Command For:**” click on **Oven**.
3. Under “**Command Name**” click on “**Turn On/Off oven**”.
4. Click on the “**Send Command**” button.

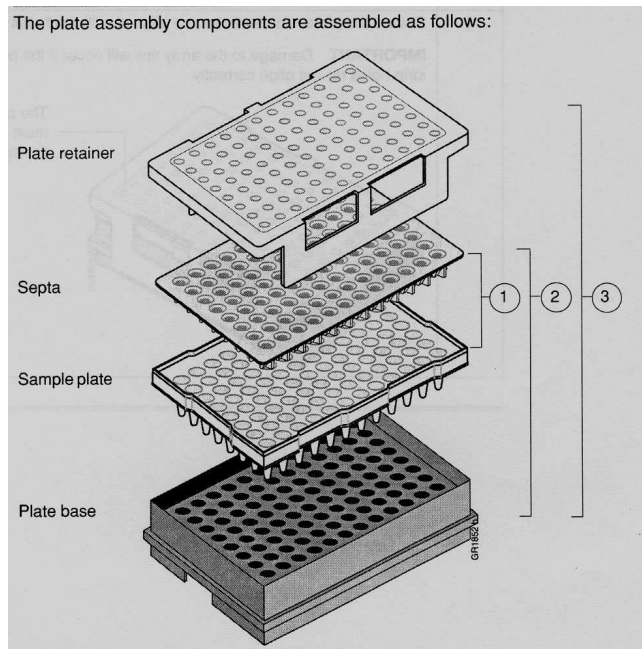


5. Under “**Command Name**” click on “**Set oven temperature**” and Under “**Value**” set it to **60**.
6. Click on the “**Send Command**” button.
7. Once denatured, place the reaction plate into the plate base. Secure the plate base and reaction plate with the plate retainer.

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IMPORTANT: Damage to the array tips will occur if the plate retainer and septa strip holes do not align correctly.

Do not write on the septa with pen, markers, sharpies, etc. Ink may cause artifacts in samples. Any unnecessary markings or debris on the septa may compromise instrument performance.

VI. Placing the Plate onto the Autosampler (Linking and Unlinking Plate)

1. In the tree pane of the Foundation Data Collection v3.0 software click on **GA Instrument** > **ga3130xl** > *instrument name* > **Run Scheduler** > **Plate View**
2. Push the tray button on the bottom left of the machine and wait for the autosampler to move forward and stop at the forward position.
3. Open the doors and place the tray onto the autosampler in the correct tray position, A or B. There is only one orientation for the plate. (The notched end faces away from you.)

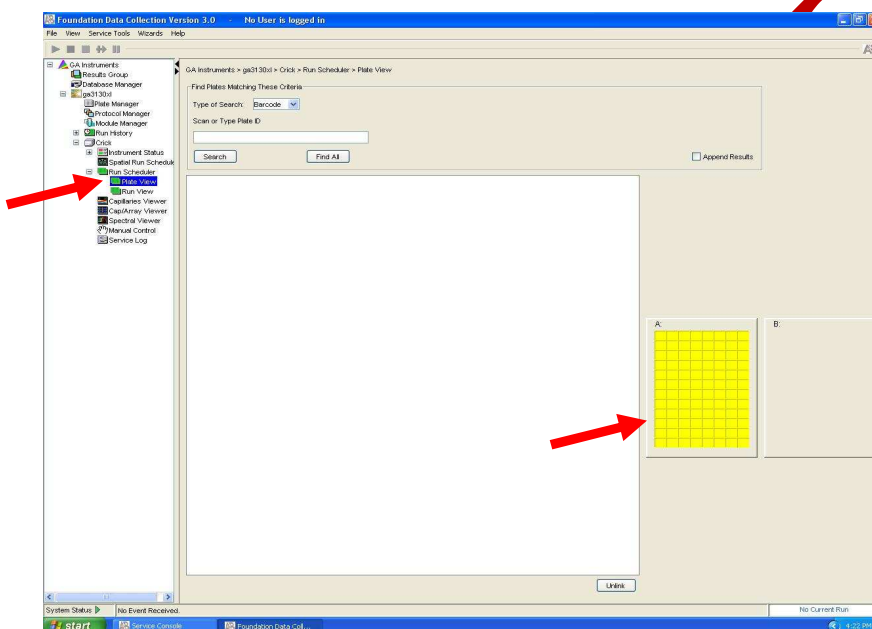
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4. Ensure the plate assembly fits flat in the autosampler. Failure to do so may allow the capillary tips to lift the plate assembly off of the autosampler.

When the plate is correctly positioned, the plate position indicator on the **Plate View** window changes from gray to yellow. Close the instrument doors and allow the autosampler to move back to the home position.



NOTE: When removing a plate from the autosampler, be careful not to hit the capillary array. Plate B is located directly under the array, so be especially careful when removing this tray.

Linking/Unlinking the Plate Record to Plate

5. Type the exact plate name in the Plate ID window and click **“Search.”**

Note: If you do not type the plate name correctly, your plate will not be found. Instead, you will be prompted to create a new plate. Click **“No”** and retype the plate name correctly.

Or, click the **“Find All”** button and select the desired plate record.

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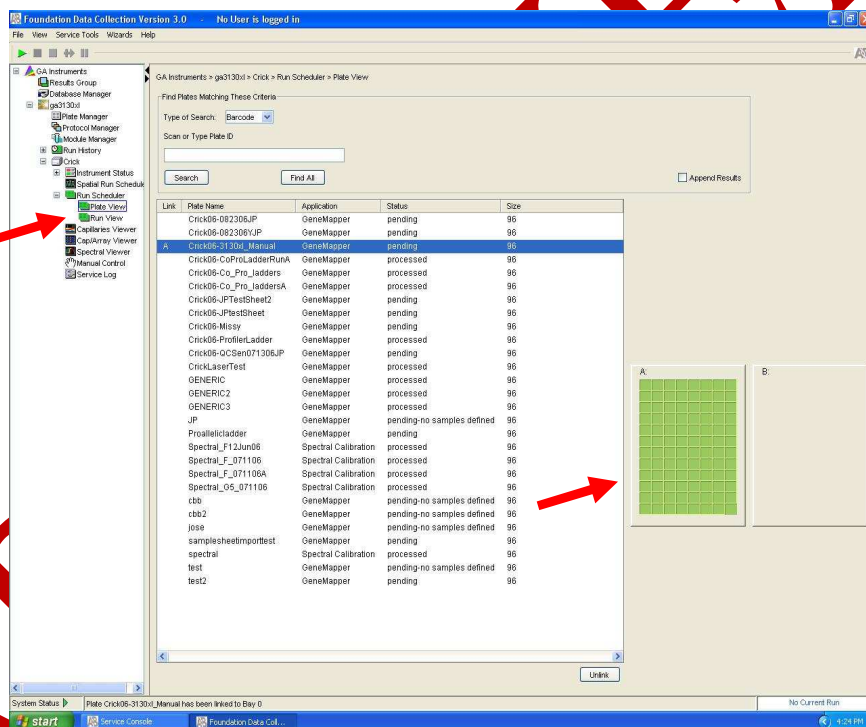
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Click the plate position indicator corresponding to the plate position in the instrument. The plate position (A or B) displays in the link column.

NOTE: It may take a minute for the plate record to link to the plate depending on the size of the sample sheet.

If two plates are being run, the order in which they are run is based on the order in which the plates were linked.

- The plate position indicator changes from yellow to green when linked correctly and the green run button becomes active.



- To unlink a plate record just click the plate record you want to unlink and click "Unlink".

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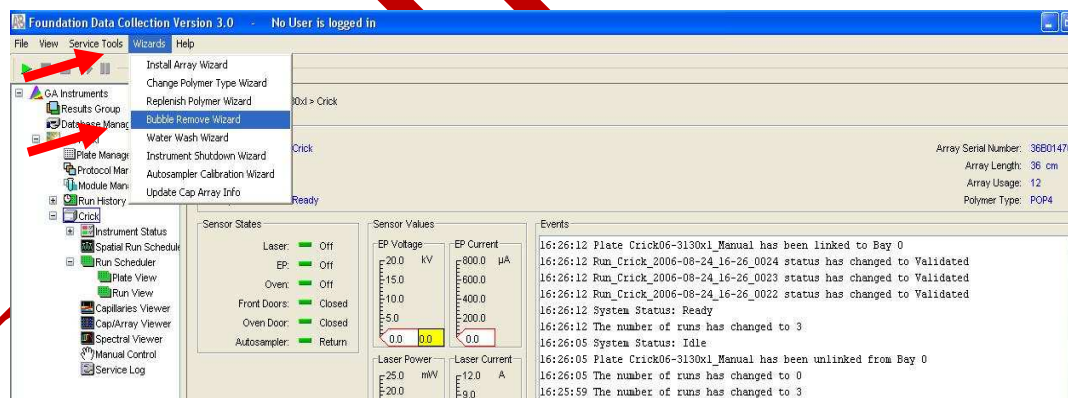
7. STR ANALYSIS ON THE ABI 3130xl GENETIC ANALYZER

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Viewing the Run Schedule

9. In the tree pane of the Foundation Data Collection software, click **GA Instruments > ga3130xl > instrument name > Run Scheduler > Run View**.
10. The **RunID** column indicates the folder number(s) associated with each injection in your run (e.g. Run_Crick_2006-08-23_11-19_0022). These folder number(s) should be recorded in the **3130XL Usage Log** binder along with the run control sheet name.
11. Click on the run file to see the Plate Map or grid diagram of your plate on the right. Check if the blue highlighted boxes correspond to the correct placement of the samples in the injections.

NOTE: Before starting a run, check for air bubbles in the polymer blocks. If bubbles are present, click on the **Wizards** tool box on the top and select **“Bubble Remove Wizard”**. Follow the wizard until all bubbles are removed.



12. Click on green **Run** button in the tool bar when you are ready to start the run. When the **Processing Plate** dialog box opens (You are about to start processing plates...), click **OK**.

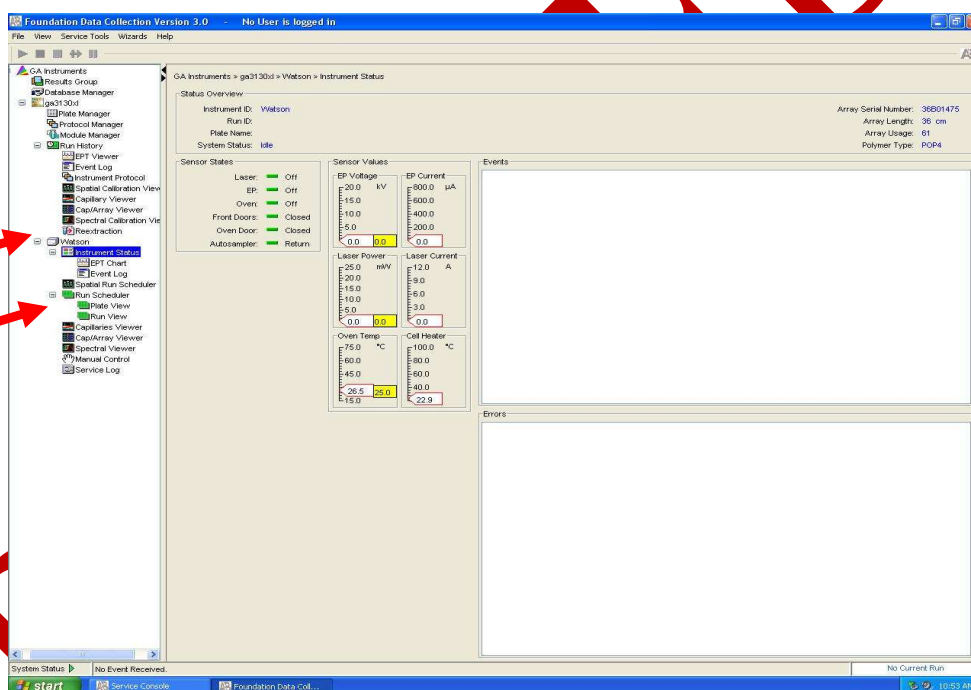
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13. To check the progress of a run, click on the **Capillary Viewer** or **Cap/ArrayViewer** in the tree pane of the Foundation Data Collection software. The **Capillary Viewer** will show you the raw data of the capillaries you select to view whereas the **Cap/Array Viewer** will show the raw data of all 16 capillaries at once.

IMPORTANT: Always exit from the **Capillary Viewer** and **Cap/Array Viewer** windows. During a run, do not leave these pages open for extended periods. This may cause unrecoverable screen update problems. Leave the **Instrument Status** window open.



The visible settings should be:

EP voltage 15kV
EP current (no set value)
Laser Power Prerun 15 mW
Laser Power During run 15mW
Laser Current (no set value)
Oven temperature 60°C

Expected values are:

EP current constant around 120 to 160µA
Laser current: 5.0A ± 1.0

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It is good practice to monitor the initial injections in order to detect problems.

	Normal Run Parameters	ReRun Parameters
Oven Temp	60°C	60°C
Pre-Run Voltage	15.0 kV	15.0 kV
Pre-Run Time	180 sec	180 sec
Injection Voltage	3 kV	5 kV
Injection Time	10 sec	20 sec
Run Voltage	15 kV	15 kV
Run Time	1500 sec	1500 sec

VII. GeneScan Analysis

When a run is complete, it will automatically be placed in the **Current Runs** folder, properly labeled with the *instrument name, date and runID* (e.g. **Run_Crick_2006-07-13-0018**).

Converting Run for GeneScan Analysis

1. On the desktop, click on the shortcut for the **RJW** conversion program.



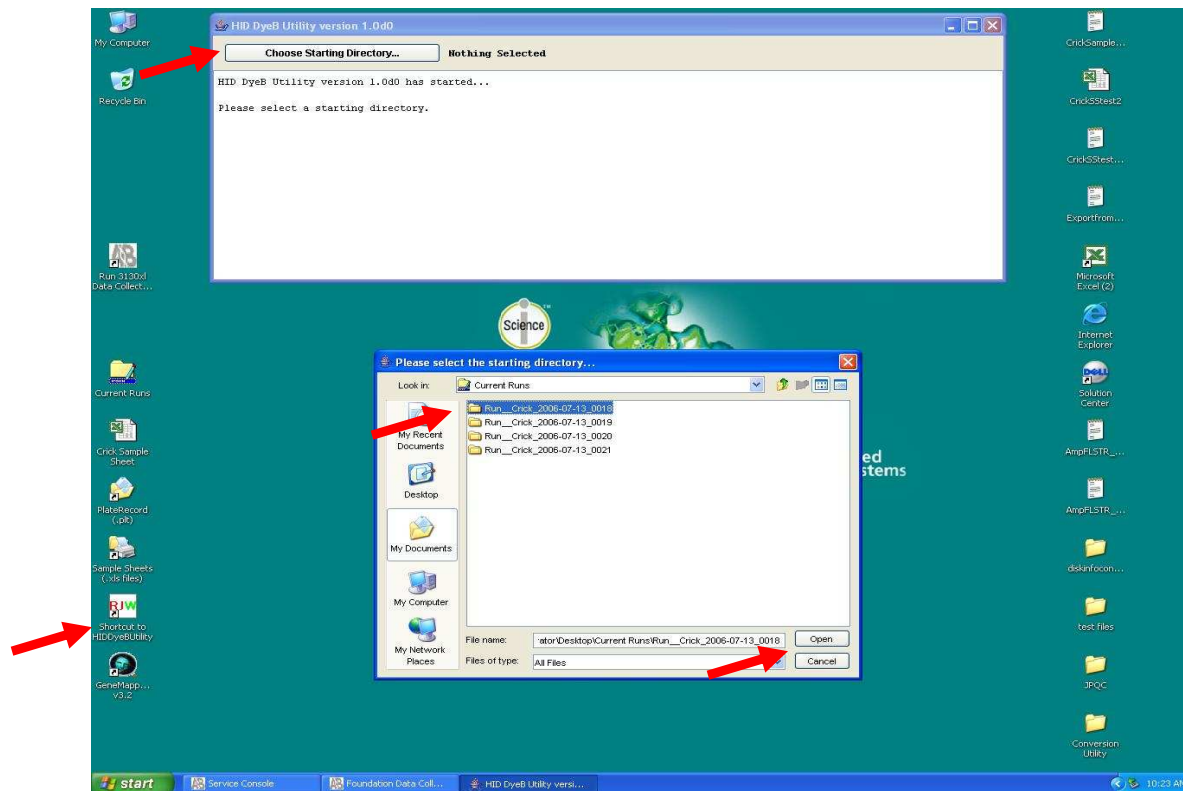
HIDDyeBUtility.exe

2. On the top of the **RJW** conversion program window click on the “**Choose Starting Directory**” button.
3. Browse the **Current Runs** Folder for your run folder(s) (e.g. **Run_Crick_2006-07-13-0018**).

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4. Double click on your run folder(s) then hit **Open**. The run folder(s) are now converted.
5. Your samples are now ready to be analyzed in **GeneScan**. Organize the run folders (there should be one for each injection) on the instrument computer into a common folder. This common folder should be named the same as your run sample sheet/plate record.
6. Transfer the common folder using any formatted CD-RWs or a Flash Drive to an Analysis computer.
7. Follow analysis procedure as outlined in [Section 8 of the Forensic Biology STR Manual v9.0](#)

NOTE: When analyzing in Genescan, the size standard column must be changed from HID Fragment Analysis to the respective system the samples were typed in.

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VIII. Water Wash and POP Change

Refer to Section I pg. 2 for schematic of 3130xl while proceeding with the water wash and pop change procedure.

1. Remove a new bottle of POP4 from the refrigerator.
2. Select **Wizards > Water Wash Wizard**
3. Click "Close Valve"
4. Open instrument doors and remove the empty POP bottle.
5. With a dampened Kimwipe® wipe the polymer supply tube and cap. Dry.
6. Replace POP bottle with the water bottle filled to the top with Gibco® Water.
7. Remove, empty, and replace the anode buffer jar on the lower polymer block.
8. Click "Water Wash." This procedure will take approximately 4 minutes.
9. When the water wash is finished click "Next"
10. Select "Same Lot" or "Different Lot"
11. Remove water bottle from the lower polymer block. Dry supply tube and cap with a Kimwipe®.
12. Replace with a new bottle of room temperature POP.
13. Click "Next."
14. Click "Flush." This will take approximately 2 minutes to complete.
15. Inspect the pump block, channels, and tubing for air bubbles.
16. Click "Next."

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17. Carefully inspect the pump blocks and pump channels for bubbles.
 - If there are bubbles click “Yes, Bubbles are present” and the “Remove bubbles” icon. Wait approximately one minute. Repeat until all bubbles are removed.
 - If bubbles are not present click “No, All bubbles are gone” and click “Next.”
18. Flush the array port by loosening the knob 1 turn **counterclockwise**.
19. Click “Flush Array Port.” Repeat if necessary.
20. When all the bubbles are gone, tighten the array port knob clockwise until it is flush against the pump block.
21. Click “Bubbles are gone” icon then “Next.”
22. Close instrument doors and wait for steady green light.
23. Click “Fill Array.” Filling the array will take approximately 2 minutes.
24. When this is completed, remove the anode buffer jar, empty, and fill with 1x TBE Buffer (~15 mL).
25. Close instrument doors and wait for the steady green light.
26. Click “Finish.”
27. Return to Section I, Step 11.

Revision History:

February 5, 2007 – This section used to be “STR Analysis on the ABI 310 Genetic Analyzer.” Entire section was replaced to detail protocols for the ABI 3130xl Genetic Analyzer. See [Approval Form](#).

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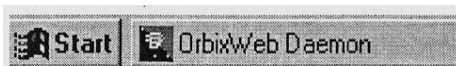
8. STR ANALYSIS ON THE ABI 3100 GENETIC ANALYZER

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Only the Cofiler, Profiler Plus, and YM1 (Y-STR) amplification systems have been validated for casework. PowerPlex 16 has been validated for Exemplar/Reference samples only, and can be run on this instrument.

How to start

Turn on the computer. Make sure computer is fully booted and the OrbixWeb Daemon is running. This is indicated by the presence of the following OrbixWeb Daemon icon and text next to the **Start** icon on the lower left portion of the desktop.



If the instrument is not on, turn it on. The status bar light will change from solid yellow (indicates instrument is booting) to blinking yellow (indicates machine is communicating with computer) and then to solid green (indicates instrument is ready for command).

NOTE: Collection cannot be launched unless the light on the instrument is green!
Important phrase to remember, “If the light isn’t green, the machine becomes mean!”

The program that is used to operate the run and collect the data is the ABI Prism 3100 Collection Software. To begin collection, double click on the **3100 Collection Software** icon.

Here is how to decide at which point of the manual to proceed:

1. If the number of injections you are running plus the capillary usage number will exceed 150 injections, proceed with **Section A “Changing and Installing the Capillary”**.
2. If the amount of POP4 in the installed syringe is low (<600 µL/tray) or greater than one week old, you will have to change it. Start with **Section C “Changing/Filling Syringes with POP4”**.
3. If the capillary and POP4 are fine at the first run of the day, start with **Section D “Changing the Buffer”** to change the buffer and water.
4. If the instrument has been run prior today and buffer and water had been changed then proceed to **Section E “Creating a Plate Record Through Excel”**.

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A. Changing and Installing the Capillary

The 3100 capillary array is a replaceable unit composed of 16 silica capillaries. The capillary usage number refers to the number of times the array has injected a set of 16 samples. Check the capillary array usage number by clicking on the **Status View** tab at the top of the collection screen. If this number, plus the number of your injection sets (6 for a 96 well plate) exceeds 150, the capillary array should be changed.

To replace a capillary array or to install a capillary array on an instrument, close the oven and instrument doors. From the **Tools** menu, select **Install Capillary Array Wizard**. Follow the directions given in the wizard to replace or install an array.

The steps involved in the **Capillary Array Wizard** are:

1. From the Capillary Array Wizard window, select the **Remove and Discard the Array** option. Click **Next**.
2. Push the tray button. Wait for the tray to stop moving before opening the instrument doors. The Capillary Array Wizard will then instruct you to “open instrument door, oven door, and detection block door”.
3. Unclip the leading-end head combs of the array from the oven by pulling on the pins to the left and right of the comb.
4. To loosen the screw holding the end of the capillary inside the upper polymer block, unscrew towards you. Remove the syringe guard and pull the upper polymer block out towards you, gently but with a bit of force, until you feel the block reach a stop point. The capillary detection cell will pop out of position. Remove the capillary from the oven slots holding it in place.
5. Completely remove the screw and ferrule holding the end of the capillary inside the upper polymer pump block. Remove the capillary assembly and discard it.
6. Completely unscrew and remove syringes and tubing from upper and lower polymer blocks. Remove upper and lower polymer blocks from instrument. Clean the upper polymer block with hot, NOT BOILING, water using the cleaning syringe. Also clean the ferrule and screw. A final rinse of deionized water should be done. Vacuum or air dry the block and put the block back in the instrument without sliding either block all the way in. Click **Next**.

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7. Select **Install a New Capillary Array** from the options. Enter the **Serial Number** of the capillary array located on the capillary array box. Also record the serial number of the capillary and the date in the 3100 Capillary Electrophoresis Log binder. Keep the capillary box on top of the instrument for future use. Click **Next**.
8. Slide the capillary leading-end head comb holder into the oven and snap the loading end of the array in place. To secure the capillary comb in place, push both buttons until they snap into place.
9. Place the capillary placement slots into their respective positions in the oven.
10. Remove the detection window protection clip from the capillary window. Make sure the window is clean (no dust or smudges). Slide the capillary assembly into the upper polymer block with the screw and ferrule and place the capillary detection cell in place. Push the polymer block back into place. Tighten the array assembly into the polymer block. Click **Next**.
11. Close the laser detection door and screw it shut. Close the oven door.
12. Fill the clean syringes with polymer and install them on the polymer block. Return the syringe guard. Click **Next**.
13. Push the pistons on both syringes slowly to remove any air bubbles. Continue to push the piston of the large syringe to fill both the upper and lower block with polymer.

IMPORTANT: Pay close attention to any air bubbles trapped next to the end of the capillary assembly and remove them. Also, make sure that all air bubbles are removed from the assembly tubing connecting the upper and lower polymer block. This is a non-transparent tube and bubbles are not visible through it. Any bubbles left in it can cause electrophoresis problems.
14. Close the instrument doors and wait for the autosampler to stop moving. Click **No** if there is no air bubbles remaining. Click **Next**.
15. Click **Fill** to fill the new capillary array. This will take a few minutes. Wait for the **Finish** button to become active, and then click it.

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16. Push the **Tray** button to present the tray. Once the autosampler has stopped moving, open the doors and change the buffer in the anode jar and the water and buffer in the reservoirs in the autosampler. Close the doors and Click **Finish**. The software will not accept the new information unless the Finish button is clicked.
17. Once the capillary array wizard is finished, a spatial calibration must be done as described in the following section.
18. Monitor the EP Current closely during the first injection. Make sure the EP Current is not fluctuating too much (ex. going from 20 to 160 then down again, etc.) Fluctuation of the EP Current is a good indicator that bubbles may be present and if the run is stopped immediately, the lower block can be saved. If, after 20 minute of the first run, the EP Current is constantly around 150 or 160, it's good.

B. Performing a Spatial Calibration

A spatial calibration provides information about the position of the fluorescence from each capillary on the CCD. It must be performed after each time a capillary array is replaced or temporarily moved from the detection block (as is done during the change of the POP4).

1. Select **Perform Spatial Calibration** from the **Tools** menu.
2. Click **Start**. The calibration will take 2 or 6 minutes (depending on if the capillaries are being filled).
3. Upon completion, a dialog box will indicate if the spatial calibration was successful. If successful, click **Details** to view the **Spatial Calibration Profile** window. Successful results yield 16 sharp peaks with similar heights having position values from 13 to 16 units (15 is optimal) higher for each subsequent capillary. A good spatial should be greater than 8000 units high.
4. If the peaks do not fit these specifications, click **OK** to return to the previous screen, press **Start** to perform another test. Follow step 3. If the spatial still does not fit the requirements, click **OK** to return to the previous screen. Select the **Fill capillaries** check box, then click **Start**.
5. If the peaks fit the specification in step 3, click **OK** to get back to the previous dialog box. Click **OK** and then **Yes** to accept the spatial calibration that was just performed.

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6. If the spatial calibration is continuously unsuccessful, see the 3100 troubleshooting section.

Proceed with section E

C. Changing/Filling Syringes with POP4

Check the amount of POP4 that remains in the reservoir syringe (the large 5.0 mL syringe on the left). For a full tray (i.e. 96 samples or 6 injections), there should be at least 600 µL of POP4 in the reservoir syringe or the run will not start. The array fill syringe (smaller 250 µL syringe on the right) will be filled automatically by the reservoir syringe when it comes time to fill the capillary array. **Never add fresh polymer to old polymer.**

Check the 3100 Capillary Electrophoresis Log binder to see when the polymer was last changed. The polymer should be changed once a week. Document the change of polymer and its lot number in the 3100 Capillary Electrophoresis Log binder.

Completely unscrew and remove both syringes and tubing (**tubing damages easily!**) from upper and lower polymer blocks. Remove upper and lower polymer blocks from instrument. Clean the upper polymer block with hot (**not boiling!**) water using the cleaning syringe. Also clean the ferrule and screw. A final rinse of deionized water should be done. Vacuum or air dry the blocks and then put them back in the instrument (**gradually slide them back into place so as not to damage the tubing connecting them**) without sliding either block all the way in.

From the **Tools** menu, select **Change Polymer Wizard**. Follow the directions given in the wizard to put fresh polymer on the instrument.

The steps involved in the **Change Polymer Wizard** are:

1. From the Polymer Wizard window, click **Close** to close the anode buffer pin valve and **Home** to home the syringes. Click **Next**.
2. Click **Same Polymer Type**, the instrument uses only POP4 polymer at this time. Click **Next**.
3. Fill both syringes with new polymer (choose the volume to fill the larger syringe with based on the anticipated instrument use for the following week). Make sure the syringes are free of any air bubbles and install both syringes on the polymer block. Click **Next**.

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4. Enter the polymer **lot number** and **expiration date**. Click **Open** to open the anode buffer. Click **Next**.
5. Push the pistons on both syringes slowly to remove any air bubbles in the block (the small syringe works best for removing bubbles stuck in the corner of the block nearest the capillary). Continue to push the piston of the large syringe to fill both the upper and lower block with polymer and insure all air bubbles have been flushed into the lower buffer chamber.

IMPORTANT: Pay close attention to any air bubbles trapped next to the end of the capillary assembly and remove them. Also, make sure that all air bubbles are removed from the assembly tubing connecting the upper and lower polymer block. This is a non-transparent tube and bubbles are not visible through it. Any bubbles left in it can cause electrophoresis problems.
6. Click **No** when all of the air bubbles are gone. Click **Next**.
7. Replace the buffer in the anode jar (see section D) and Click **Finish**.
8. The capillary must be flushed of old POP4 with new POP4. From **Instrument** in the main menu, select **Manual Control**. Under the Command category select **Capillary**, in Command name select **Fill**, under value select **36cm**.
9. Monitor the EP Current (displayed in status window) closely during the first injection. Check and make sure the EP Current is not fluctuating too much (ex. going from 20 to 160 then down again, etc.) Fluctuation of the EP Current is a good indicator that bubbles may be present and, if the run is stopped immediately, the lower block can be saved. If, after 20 minute of the first run, the EP Current is constantly around 150 or 160, it's within the normal range.

NOTE: You must change the polymer weekly. The polymer is good at 25°C for about 7 days.

ATTENTION: IF the pump block was removed and the capillary window was disturbed, you must perform a spatial calibration (see B.) before running samples.

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D. Changing the Buffer

The cathode buffer and water reservoirs located on the autosampler tray and anode buffer jar located below the lower pump block should be changed at the beginning of each day the instrument is in use.

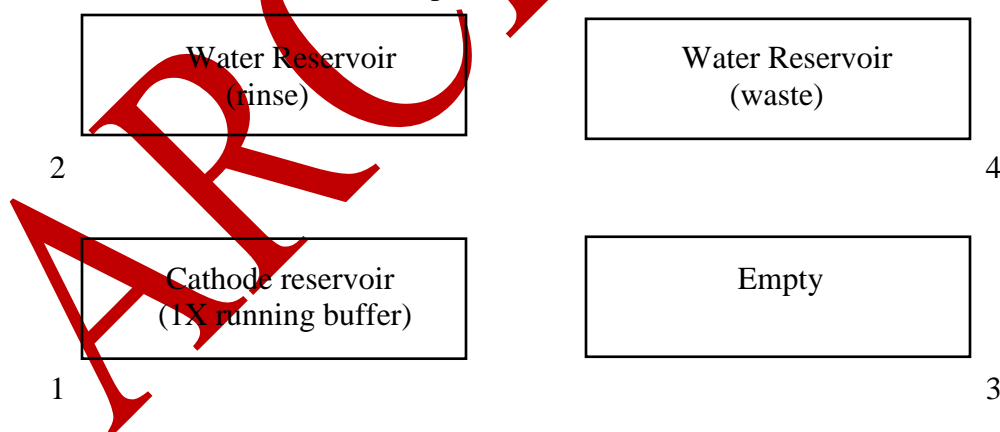
1. To fill the water and cathode buffer reservoirs on the autosampler, close the instrument doors and press the tray button on the outside of the instrument to bring the autosampler to the forward position. **Wait until the autosampler has stopped moving and then open the instrument doors.**
2. Remove all reservoirs and anode jar and dispose of the remaining fluid. Rinse and fill the water reservoirs to the line with deionized water and the cathode reservoir and anode with 1X running buffer, about 16mL each. Dry the outside and inside rim of the reservoirs using a Kimwipe and place a clean septa strip on each reservoir, as needed.

Place the reservoirs in the instrument in their respective positions, as shown below.

3. Close the instrument doors.

NOTE: When using the 3100 for fragment analysis, it is not necessary to fill the water reservoir in position #3. This reservoir is used only for DNA sequencing applications.

Position of reservoirs in autosampler:



NOTE: Be sure that the septa fit snugly and flush on the tops of the reservoirs in order to prevent damage of the capillary tip. Also, it is important that there is no condensation on the inside of the reservoir. The septa strips must be dry!

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E. Creating a Plate Record Through Excel

This excel sheet will be imported into the 3100 collection software.

1. Double click on the **Sample Sheet** icon on the desktop to open the sample sheet template.
2. Make sure that the “Sample Sheet” tab is selected (bottom left of the screen).
3. Fill in the sample sheet name in cell E3.

IMPORTANT: When naming a plate/sample sheet, you may use letters, numbers, and the following characters only: -_(){}#.+ . Also, no spaces between characters are allowed. If an incorrect character is used in the sample sheet name, you will not be allowed to import the plate record into the 3100 collection software.

Casework sample sheets should be named indicating the instrument, the year and the consecutive run number for the multiplex, e.g.

Stars04-021Co-009Pro or Rudy04-004Co or Stripes04-015Y

4. Fill in the sample information in the **case number-sample description-tube label** columns. Room has already been reserved for the Allelic Ladders in wells A1, A3, A5, A7, A9, A11 (the grey cells) and the positive control in well B1 (the positive control may be edited, though the ladders may not, as it is mandatory that there be a ladder included with each injection set).

IMPORTANT: As a result of a bug in Excel, the cut and paste function should not be used. Instead, highlight the cell(s) you wish to move and press <ctrl-c>. Right click on the first destination cell and choose “Paste Special...”. Under “Paste” select “Values” and click OK.

5. In the “Sys.” column, fill in the appropriate letter for the system in which the sample was amplified: ‘P’ for Profiler Plus, ‘C’ for Cofiler, ‘I’ for Identifiler, ‘M’ for YM1, and ‘X’ for PowerpleX 16. For reruns, use: ‘PR’ for Profiler Plus Reruns, ‘CR’ for Cofiler Reruns, ‘IR’ for Identifiler Reruns, ‘MR’ for YM1 Reruns, and ‘XR’ for PowerpleX 16 Reruns.

IMPORTANT: If you do not specify the system, sample data will not be collected. If you fill in the wrong system, sample data will be collected incorrectly.

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IMPORTANT: The 3100 uses the same injection conditions (time/voltage) for all 16 samples in a given injection. Therefore, samples that need to be run with non-standard conditions (such as most reruns) cannot be part of the same injection as normally run samples.

6. Save the sample sheet by selecting **Save As** from the File menu. Save in D:\appliedbio\3100\Bin\Sample Sheets (.xls files). This way you will have the file backed up in case further changes or additions are necessary.
7. If the sample sheet is full and/or you are ready to run the plate, print it out. If you only have filled out the first page, please set the printer to print only from page 1 to page 1.
8. Click on the **Second Tab** labeled “SaveAs.plt” or “test” on the bottom left of the screen. Select **Save As** from the File menu and do the following:
 - Change the file directory to D:\appliedbio\3100\Bin\PlateRecords(.plt).
 - Change **Save as file type** to “text(Tab-delimited)(*.txt)”.
 - Change the file name so that its extension is **.plt** and there are no spaces, the file name should be of the format: *yourfilename.plt*
9. Quit Excel. You will be prompted to save again by Excel, this is not necessary so select **NO**.
10. Open the 3100 Collection software. Select the **Plate View**. Click on the **Import** button. Open the Plate Records (.plt) folder and select your plate record file. Click **OK**.

F. Preparing and Running the DNA Samples

Before preparing the samples, turn on the oven. This saves time. The oven would turn on automatically after a run is started, but in order to monitor the first injection one would have to wait for the oven to reach 60°C.

1. Under **Instrument**, select **Manual Control**. Under the **Command category** select **Oven**. Make sure the **Set State** is selected under **Command Name** and under **Value** turn the oven **On**. Click **Send Command**.
2. Now change the **Set State** button to **Set Temperature** and under **Value** enter **60**. The oven will not heat unless it is turned on first, then the temperature can be set. Click on **Send Command**.

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3. Arrange amplified samples in a 96-well rack according to how they will be loaded into the 96- well reaction plate. Sample order is as follows: A1, B1, C1, D1... G1, H1, A2, B2, C2... G2, H2, A3, B3, C3, etc. Thus the plate is loaded in a columnar manner where the first injection corresponds to wells A1-H2, the second A3-H4 and so on. Remember that an allelic ladder is run as the first sample of each injection. Therefore, leave room for the allelic ladders in A1, A3, A5, etc.

Label the side of the reaction plate with the name used for the Plate Record with a sharpie and place the plate in the plate base (see diagram on page 102).

For Cofiler and Profiler:

4. For N+2 samples mix 9.5 μL of HiDi Formamide with 0.5 μL of GS500 Rox Standard per sample. Mix thoroughly by vortexing.

Use the following volumes for the necessary number of samples:

# Samples +2	HiDi Formamide	GS500 Standard
18	171 μL	9 μL
34	323 μL	17 μL
50	475 μL	25 μL
66	627 μL	33 μL
82	779 μL	41 μL
98	931 μL	49 μL
114	1083 μL	57 μL
130	1235 μL	65 μL

5. Aliquot 10 μL of the formamide/standard mixture into each well being used on the 96-well reaction plate.

IMPORTANT: If one injection has less than 16 samples add 12ul of either dH₂O or buffer or formamide/standard mix to all unused wells.

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Have someone witness the tube setup by comparing the tube labels and positions indicated on the sample sheet with the tube labels and positions of the tubes themselves (tubes should be laid out to correspond with their respective positions in the 96 plate). Add 1 μL of allelic ladder to the respective tube(s). Add 2 μL of PCR product to each tube according to the sample sheet.

When samples are being rerun to obtain more information, use 4 μL of PCR product and 4 μL of a 1/10 dilution of the positive control and allelic ladder.

IMPORTANT NOTES FOR RE-RUN SAMPLES:

1. Re-runs “high” **cannot** be on the same injection as non-re-run samples.
2. Re-runs “high” **must always** be the last injection(s) of the run and **must always** have positive controls and allelic ladders run under the same “high” conditions.
3. Re-runs “normal” may be integrated with non-re-run samples.

When adding PCR product, make sure to pipet the solution directly into the formamide and gently flush the pipet tip up and down a few times to mix it.

Proceed to step 6.

For PowerPlex 16:

4. For N+2 samples mix 9 μL of HiDi Formamide with 1.0 μL of ILS600 Size Standard per sample. Mix thoroughly by vortexing.

Use the following volumes for the necessary number of samples:

# Samples +2	HiDi Formamide	ILS600 Standard
18	162 μL	18 μL
34	306 μL	34 μL
50	450 μL	50 μL
66	594 μL	66 μL
82	738 μL	82 μL
98	882 μL	98 μL
114	1026 μL	114 μL
130	1170 μL	130 μL

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You might need more than N+2 if you are working with microtiter plates and a multichannel pipet.

Aliquot 10 µL of the formamide/size standard mixture into each well being used on the 96-well reaction plate.

IMPORTANT: If one injection has less than 16 samples add 12µl of either dH₂O or buffer or formamide/standard mix to all unused wells.

Have someone witness the tube setup by comparing the tube labels and positions indicated on the sample sheet with the tube labels and positions of the tubes themselves (tubes should be laid out to correspond with their respective positions in the 96 plate). If you are aliquoting from a 96 well tray, you do not need a witness. Add 1 µL of allelic ladder to the first well of each injection. Add 2 µL of PCR product to each tube according to the sample sheet.

When samples are being rerun to obtain more information, use 4 µL of PCR product and 4 µL of a 1/10 dilution of the positive control and allelic ladder.

IMPORTANT NOTES FOR RE-RUN SAMPLES:

1. Re-runs “high” **cannot** be on the same injection as non-re-run samples.
2. Re-runs “high” **must always** be the last injection(s) of the run and **must always** have positive controls and allelic ladders run under the same “high” conditions.
3. Re-runs “normal” may be integrated with non-re-run samples.

When adding PCR product, make sure to pipet the solution directly into the formamide and gently flush the pipet tip up and down a few times to mix it.

Proceed to step 6.

For YM1 STRs:

4. For N+2 samples mix 9.5 µL of HiDi Formamide with 0.5 µL of LIZ Size Standard per sample. Mix thoroughly by vortexing. Use the following volumes for the necessary number of samples:

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# Samples +2	HiDi Formamide	LIZ Standard
18	171 μ L	9 μ L
34	323 μ L	17 μ L
50	475 μ L	25 μ L
66	627 μ L	33 μ L
82	779 μ L	41 μ L
98	931 μ L	49 μ L
114	1083 μ L	57 μ L
130	1235 μ L	65 μ L

5. Aliquot 10 μ L of the formamide/standard mixture into each well being used on the 96-well reaction plate.

IMPORTANT: If one injection has less than 16 samples add 12ul of either dH₂O or buffer or formamide/standard mix to all unused wells.

Have someone witness the tube setup by comparing the tube labels and positions indicated on the sample sheet with the tube labels and positions of the tubes themselves (tubes should be laid out to correspond with their respective positions in the 96 plate). Add 2 μ L of PCR product to each tube according to the sample sheet.

When samples are being rerun to obtain more information, use 4 μ L of PCR product and 4 μ L of a 1/10 dilution of the positive control and allelic ladder.

IMPORTANT NOTES FOR RE-RUN SAMPLES:

1. Re-runs "high" **cannot** be on the same injection as non-re-run samples.
2. Re-runs "high" **must always** be the last injection(s) of the run and **must always** have positive controls and allelic ladders run under the same "high" conditions.
3. Re-runs "normal" may be integrated with non-re-run samples.

When adding PCR product, make sure to pipette the solution directly into the formamide and gently flush the pipette tip up and down a few times to mix it.

Proceed to step 6.

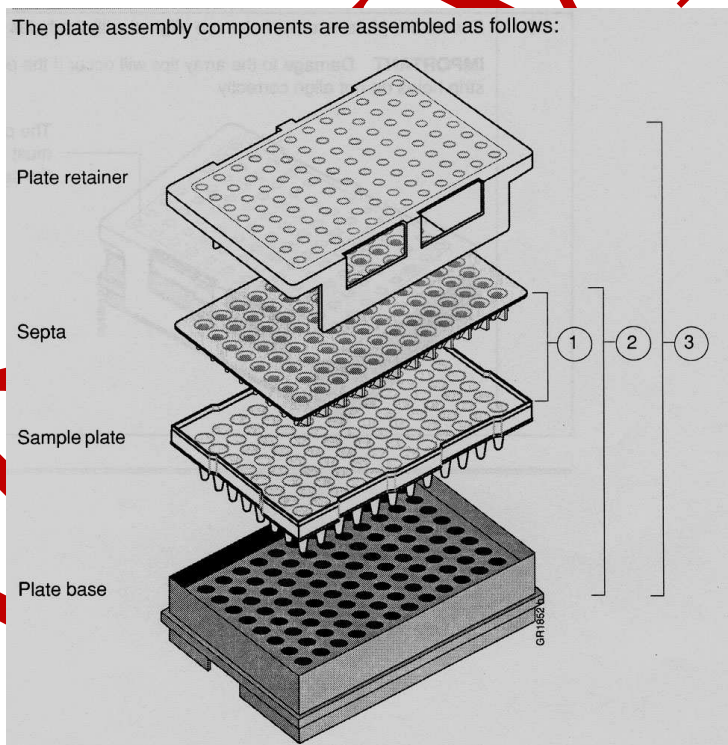
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All Multiplexes:

- Once all of the samples have been added to the master mix, place a new 96-well Septa over the reaction plate and firmly press the septa into place. Spin plate in centrifuge for one minute.
- Remove the reaction plate from the base and heat denature samples at 95°C for 5 minutes followed by a quick chill at 4°C for 5 minutes using the denature/chill program on an ABI 9700 thermal cycler. **Make sure to keep the thermal cycler lid off of the sample tray to prevent the septa from heating up.** After removing tray from the heat block, check the wells for air bubbles. If there are any, centrifuge the tray to remove any air bubbles.
- Once denatured, place the reaction plate into the plate base. Secure the plate base and reaction plate with the plate retainer.



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IMPORTANT: Damage to the array tips will occur if the plate retainer and septa strip holes do not align correctly.

Do not write on the septa with pen, markers, sharpies, etc. Ink may cause artifacts in samples. Any unnecessary markings or debris on the septa may compromise instrument performance.

Placing the Plate onto the Autosampler

The Autosampler holds up to two, 96-well plates in tray positions A and B. To place the plate assembly on the autosampler, there is only one orientation for the plate, with the notched end of the plate base away from you.

1. Push the tray button on the bottom left of the machine and wait for the autosampler to move forward and stop at the forward position.
2. Open the doors and place the tray onto the autosampler in the correct tray position, A or B.
3. Ensure the plate assembly fits flat in the autosampler. Failure to do so may allow the capillary tips to lift the plate assembly off of the autosampler.

When the plate is correctly positioned, the plate position indicator on the **Plate View** page changes from gray to yellow. Close the instrument doors and allow the autosampler to move back to the home position.

NOTE: When removing a plate from the autosampler, be careful not to hit the capillary array. Plate B is located directly under the array, so be especially careful when removing this tray.

Linking the Plate Record to a Plate

1. On the **Plate View** screen, click on the plate record in the **Pending Plate Record** table that you are linking. Then click the plate position (A or B) that corresponds to the plate you are linking.

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2. Once the plate has been linked, the:
- **Run Instrument** button on the toolbar becomes highlighted, meaning that the instrument is ready to run.
 - Plate position indicator for the linked plate changes from yellow to green
 - Plate record moves from the **Pending Plate Records** table to the **Linked Plate Records** table

NOTE: It may take a minute for the plate record to link to the plate depending on the size of the sample sheet.

If two plates are being run, the order in which they are run is based on the order in which the plates were linked.

3. Click the **Run View** tab to view the run schedule. The **RunID** column indicates the folder number(s) associated with each injection in your run. These folder number(s) should be recorded in the **3100 Log** binder along with the run control sheet name. Click **Run** to start the run. NOTE: Before starting a run, check for air bubbles in the polymer blocks.

To check the progress of a run, click on the **Array View** or **Capillary View** tab at the top of the collection screen. The **Array View** window will show the raw data of all 16 capillaries at once. The **Capillary View** window will show you the raw data of the capillaries you select to view.

IMPORTANT: Always exit from the **Array View** and the **Capillary View** windows. During a run, do not leave these pages open for extended periods. This may cause unrecoverable screen update problems. Leave the **Status View** window open.

The visible settings should be:

EP voltage 15kV
EP current (no set value)
Laser Power Prerun 7.5mW
Laser Power During run 15mW
Laser Current (no set value)
Oven temperature 60°C

Expected values are:

EP current constant around 120 to 150µA
Laser current: 4.5A

It is good practice to monitor the initial injections in order to detect problems.

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Unlinking a Plate:

1. In the **Linked Plate Records** table of the **Plate View** page, select the plate record that you want to unlink.
2. Click **Unlink**. The plate record will return to the Pending Plate Record table and the plate position indicator will return to yellow.

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When a run is complete, it will automatically be placed in the current run folder, properly labeled with the *instrument name, date and runID* (e.g. **Run_3100_Stars_2004-1-31-85**). Before processing the samples, organize the folders for each injection into one common folder. This folder should be named the same as your run sample sheet/plate record. NOTE: If a run has 6 injections, you will have 6 folders for that run.

To view the data from the run, open the 3100 GeneScan Analysis Software for Windows NT by double-clicking on the **GeneScan** icon on the desktop. The analysis program will launch.

Creating A New Project

To create a new project, under **File** select **New**. The **Create New** dialog box appears.

1. Click the **Project** icon. An untitled **Analysis Control** window opens.
2. To add sample files to the open analysis control window, click on **Project** from the menu options and select **Add Sample Files**.
3. When the **Add Sample Files** dialog window appears, find the **Current Run** folder containing the injection folders with the samples that you want to add to the project. Find the specific injection folder(s) that you have previously documented on your sample sheet and open by double-clicking on the folder icon of interest. At this point you will see icons representing each individual sample, all belonging to one injection.

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To add samples to a project, take the following action:

If you want to...	Then...
Select a single sample file	Double-click the file OR select the file and click Add
Select all the sample files	Click Add All
Add a continuous list of sample files	a. Click the first sample that you want to add. b. Press the Shift key and click the last sample you want to add. Click Add . All the files between the first and last file are selected.
Add a discontinuous list of samples	a. Click the first sample that you want to add b. Press the Control key and then click on the other sample(s) you want to add. Click Add . All the files you selected will be highlighted and selected.

- Click **Finish** when you have added all of the samples.

Project File Analysis

The **Analysis Control** window shows in separate columns the dye lanes, sample file names, size standard options, and analysis parameters to choose for each lane. Boxes for the red dye lane should be marked with diamonds to indicate that red is the color for the size standard.

The samples have been auto analyzed and the matrix was installed during collection. The 3100 GeneScan Software does not place check marks in the dye lane boxes to indicate that the sample has been analyzed. Normally it is not necessary to reanalyze the samples.

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The auto analysis should have been performed using the following predefined files:

System	Size Standard File	Analysis Parameter File
Cofiler	75-450.szs	GS500Analysis.gsp
Profiler Plus	75-450.szs	GS500Analysis.gsp
PowerPlex 16	ILS600.szs	ILS600.gsp
YM1	Ystr.szs	YM1.gsp

NOTE: A new or different matrix cannot be installed on sample files after a run has been completed because the matrix is applied to the samples during sample collection and becomes an integral part of the data. Although the 3100 Genescan Software for Windows NT has the option to “Install New Matrix”, once the samples have been run a new matrix cannot be applied. If samples need a new matrix, a new spectral calibration (matrix) must be generated and the samples must be rerun using the new matrix.

To ensure that all the sizing results are correct, check the labeling of the size standard peaks for each sample.

- To view the analysis results, select **Windows** from the main menu and click on **Results Control**. The analyzed colors for each lane are shown in dark grey. The white squares mean that this color has not been analyzed.
- The raw data can be seen in up to 8 display panels, by changing the **# of panels** to **8**. To view each color separately, check **Quick Tile** to **On**.
- Select the first 8 red dye lanes by clicking on them and then click **Display**. Each sample standard will be displayed in its own window. To view all 8 standards, you must scroll through all of the windows. Continue checking your size standard for the entire tray by going back to the **Results Control** window, clicking on **Clear All** and selecting the next 8 samples. Repeat these steps until all of the sample size standards have been checked.

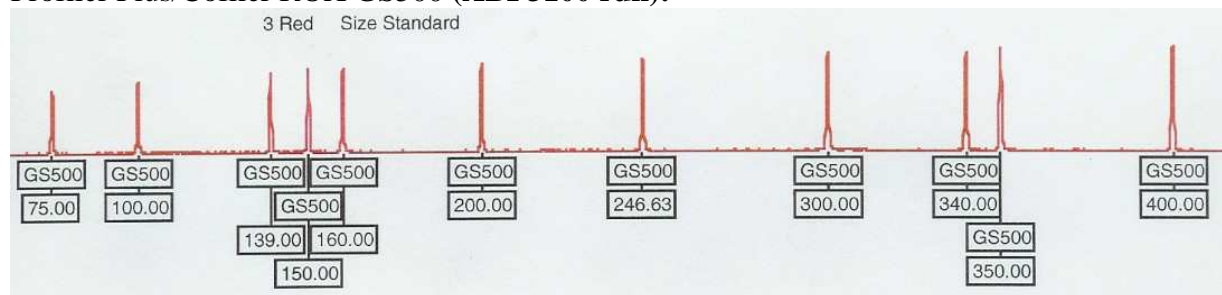
IMPORTANT: For ABI 3100 runs, the 250bp fragment in GS500 may not be labeled as 250. However, for ILS600 and CXR standards (on the ABI 310), the 250bp fragment must be labeled as such.

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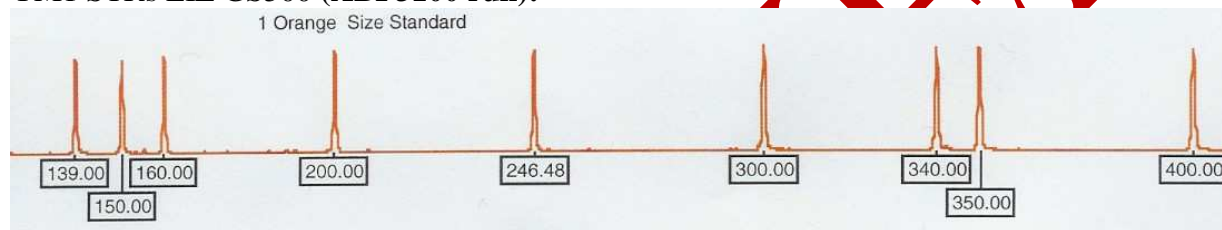
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Profiler Plus/Cofiler ROX GS500 (ABI 3100 run):

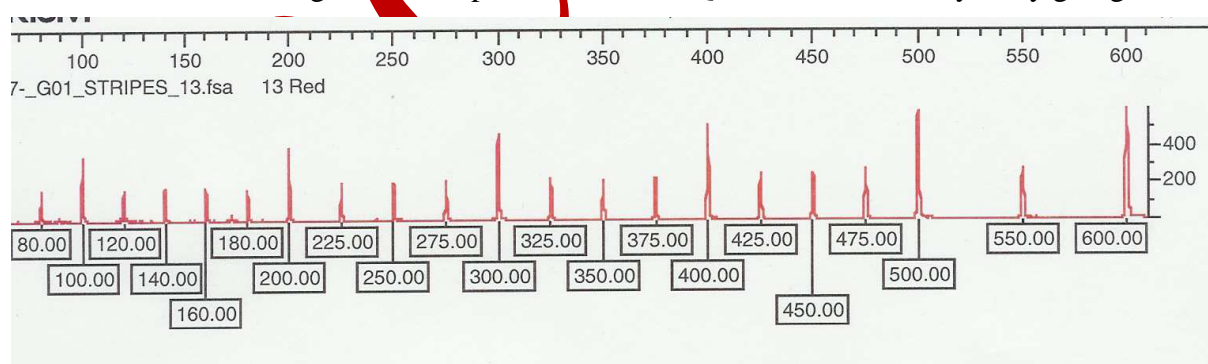


YM1 STRs LIZ GS500 (ABI 3100 run):



PowerPlex16 ILS600:

Before proceeding with the Genotyper analysis, under **File** select **Save Project As**. The project will be named according to the Sample Sheet name. Quit GeneScan Analysis by going to **File**



and Selecting **Exit**.

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Analysis Problems

1. Size standard peaks incorrect – redefine size standard

From the **Analysis control window**, go to the **Size Standard** column and click on the small arrow on the right side of the cell of one of your samples, click on it and go to **Define New**.

From here a window displaying that samples size standard will appear. Label all necessary peaks as shown above.

For ABI 3100 runs, skip the 250bp peak since it may not be labeled as 250. If you label this peak with a value, your analysis will FAIL.

When you are done defining the new size standard, click the “X” in the upper right corner of the box and save the new standard that you defined by clicking on “Save”. Name the size standard whatever you wish. Select this size standard for the analysis of all the failed samples.

2. Samples were not analyzed

If the auto-analysis did not work for the whole sample set it is possible that auto analysis was not selected before the collection or that the wrong .gsp file was attached. In the sample list, go to the **Parameter** drop down menu (click on arrow) and select “**GS500analysis.gsp**” for Cofiler and Profiler Plus or “**ILS600.gsp**” for Powerplex 16. Reanalyze samples.

It is also possible, that the run was either too fast or too slow. The analysis range may need to be changed. Look at raw data by highlighting a sample and under **Sample** choose **Raw data**.

Under **Settings** go to **Analysis Parameters**. The default analysis range for Profiler Plus and Cofiler is 2300 to 7200 data points (see picture below), for Powerplex 16 the setting is 1800 to 10000 data points.

Extend the range by choosing an earlier start point, e.g. 2100. Close window. In the sample list, go to the **Parameter** drop down menu (click on arrow) and select “**Analysis Parameters**” instead of the default GS500analysis.gsp. Reanalyze samples.

Do not change the settings in the GS500analysis.gsp parameter file; do not create any new parameter files.

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The screenshot displays the 'GS500Analysis.gsp' window with the following settings:

- Analysis Range:** ☒ This Range (Data Points). Start: 2300, Stop: 2200.
- Size Call Range:** ☒ This Range (Base Pairs). Min: 75, Max: 400.
- Data Processing:**
 - Smooth Options:** ☒ Light.
 - Size Calling Method:** ☒ Local Southern Method.
- Peak Detection:**
 - Peak Amplitude Thresholds:** B: 75, Y: 75, G: 75, R: 75.
 - Min. Peak Half Width:** 2 Pts.
 - Polynomial Degree:** 3.
 - Peak Window Size:** 10 Pts.
 - Slope Threshold for Peak Start:** 0.0.
 - Slope Threshold for Peak End:** 0.0.
- Baselining:** **BaseLine Window Size:** 251 Pts.
- Auto Analysis Only:** **Size Standard:** 75-450.szz.

3100 Analysis default

Do not change any of the settings except the range or the peak amplitude threshold for Red.

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The screenshot shows the 'YM1.gsp' window of the ABI 3100 Genetic Analyzer software. The interface is divided into several sections:

- Analysis Range:** Radio buttons for 'Full Range' and 'This Range (Data Points)'. The 'This Range' option is selected, with 'Start' set to 2700 and 'Stop' set to 7200.
- Size Call Range:** Radio buttons for 'Full Range' and 'This Range (Base Pairs)'. The 'This Range' option is selected, with 'Min' set to 100 and 'Max' set to 450.
- Data Processing:** A sub-section titled 'Smooth Options' with radio buttons for 'None', 'Light', and 'Heavy'. 'Light' is selected.
- Size Calling Method:** Radio buttons for '2nd Order Least Squares', '3rd Order Least Squares', 'Cubic Spline Interpolation', 'Local Southern Method', and 'Global Southern Method'. 'Local Southern Method' is selected.
- Peak Detection:** A section titled 'Peak Amplitude Thresholds' with input fields for 'B: 75', 'Y: 75', 'G: 75', and 'R: 75'. Below these are 'Min. Peak Half Width: 2 Pts', 'Polynomial Degree: 3', 'Peak Window Size: 19 Pts', and two 'Slope Threshold for Peak Start' and 'Peak End' fields, both set to 0.0.
- Baselining:** A section titled 'BaseLine Window Size' with an input field set to 251 Pts.
- Auto Analysis Only:** A section titled 'Size Standard:' with a dropdown menu showing 'Ystr.szs'.

3100 Analysis default

Do not change any of the settings except the range or the peak amplitude threshold for Orange (O).

3. Samples still didn't analyze

The error message for a failed analysis is: Analysis failed on Dye B, G, Y, R. Repeat the above choosing another scan range. Another possible problem is missing or too low red size standard. Look at **Raw Data** to find out. Access **Analysis Parameters** and lower threshold for red (R) to 25. Reanalyze samples.

ATTENTION: all re-analysis results and parameter changes are automatically written to the individual sample files, even if the changes to the project are not saved. Do not re-analyze casework data without a reason.

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3100 Genetic Analyzer Troubleshooting

Instrument Startup

Observation	Possible Cause	Recommended Action
No communication between the instrument and the computer (yellow light is blinking).	Instrument not started up correctly.	Make sure the oven door is closed and locked and the front doors are closed properly. If everything is closed properly, start up in the following sequence: a. Log out of the computer b. Turn off the instrument. c. Boot up the computer. d. After the computer has booted completely, turn the instrument on. Wait for the green status light to come on. e. OrbixWeb Daemon should be launched, if not already. f. Launch Data Collection software
Red light is blinking	Incorrect start up procedure	Start up in the following sequence: a. Log out of the computer. b. Turn off the instrument. c. Boot up the computer. d. After the computer has booted completely, turn the instrument on. Wait for the green status light to come on. e. OrbixWeb Daemon should be launched, if not already. f. Launch the Data Collection Software.
Data Collection software will not launch.	Did not launch Orbixweb Daemon first .	Relaunch application following OrbixWeb Daemon

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Observation	Possible Cause	Recommended Action
Computer screen is frozen	Communication error. This may be due to leaving the user interface in the Capillary View or Array View window.	There will be no loss of data. However, if the instrument is in the middle of a run, wait for the run to stop. Then, exit the Data Collection software and restart as described above.
Autosampler does not move to the forward position.	Possible communication error OR Oven or instrument door is not closed.	Restart the system, and then press the Tray button. OR a. close and lock the oven door. b. close the instrument doors. c. Press the Tray button.
Communication within the computer is slow.	Database is full.	Old files need to be cleaned out of the database. Follow proper manual procedures described in the ABI Prism 3100 Genetic Analyzer User's Manual.

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Spatial Calibration

Observation	Possible Cause	Recommended Action
Unusual peaks or a flat line for the spatial calibration.	1. The instrument may need more time to reach stability. An unstable instrument can cause a flat line with no peaks in the spatial view. 2. Improper installation of the detection window. 3. Broken capillary resulting in a bad polymer fill. 4. Dirty detection window.	1. Check or repeat spatial calibration. 2. Reinstall the detection window and make sure it fits in the proper position. 3. Check for a broken capillary, particularly in the detection window area. If necessary, replace the capillary array using the Install Array Wizard. 4. Place a drop of METHANOL onto the detection window, and dry. Use only light air force.
Persistently bad spatial calibration results.	Bad capillary array.	Replace the capillary array, and then repeat the calibration. Call Technical Support if the results do not improve.

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Spectral Calibration

Observation	Possible Cause	Recommended Action
No signal.	1. Incorrect preparation of sample. 2. Air bubbles in sample tray.	1. Replace samples with fresh samples prepared with fresh formamide. 2. Centrifuge samples to remove air bubbles.
If the spectral calibration fails, or if a message displays “No candidate spectral files found”.	1. Clogged capillary 2. Incorrect parameter files and/or run modules selected. 3. Insufficient filling of array. 4. Expired matrix standards	1. Refill the capillaries using manual control. Look for clogged capillaries during capillary fill on the cathode side. 2. Correct the files and rerun the calibration. 3. Check for broken capillaries and refill the capillary array. 4. Check the expiration date and storage conditions of the matrix standards. If necessary, replace with a fresh lot.
Spike in the data.	1. Expired polymer. 2. Air bubbles, especially in the polymer block tubing assembly. 3. Possible contaminant or crystal deposits in the polymer.	1. Replace the polymer with fresh lot using the change Polymer Wizard. 2. Refill the capillaries using manual control. 3. Properly bring the polymer to room temperature; do not heat to thaw rapidly. Swirl to dissolve any solids. Replace the polymer if it has expired.

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Data collection software problem

Observation	Possible Cause	Recommended Action
Cannot access injection folder, error message "Autoextraction failed"	AE server not active	Start AE server by going to Windows Start, Start up folder. Close Collection software. Select: Start > Applied Biosystems > 3100 Utilities > Extractor Utility. Select the run to extract. Click Extract. The data will be extracted to the default location.

Run Performance

Observation	Possible Cause	Recommended Action
No data in all capillaries	Bubbles in the system.	Visually inspect the polymer block and the syringes for bubbles. Remove any bubbles using the Change Polymer Wizard. If bubbles still persist, perform the following: a. Remove the capillary array b. Clean out the polymer block and syringes. c. Replace polymer with fresh polymer. Make sure to draw the polymer into the syringe very slowly.
No signal.	1. Dead space at bottom of sample tube. 2. Bent capillary array. 3. Failed reaction. 4. Cracked or broken capillary	1. Centrifuge the sample tray. 2. Replace the capillary array 3. Repeat reaction. 4. Visually inspect the capillary array including the detector window area for signs of breakage.

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Observation	Possible Cause	Recommended Action
Low signal strength.	<ol style="list-style-type: none">1. Poor quality formamide.2. Pipetting error, not enough sample.3. Sample has high salt concentration.4. Insufficient Mixing5. Weak amplification of DNA	<ol style="list-style-type: none">1. Use a fresh lot of formamide2. Increase the amount of DNA added and check pipet calibration.3. Dilute in high quality water, microcon.4. Vortex the sample thoroughly, and then centrifuge the tube to condense the sample.5. Re-amplify the DNA
Elevated baseline	<ol style="list-style-type: none">1. Possible contamination in the polymer path.2. Possible contaminant or crystal deposits in the polymer.3. Poor spectral calibration.4. Detection cell is dirty	<ol style="list-style-type: none">1. Wash the polymer block with hot water. Pay particular attention to the upper polymer block, the ferrule, the ferrule screw, and the peek tubing. Dry the parts by vacuum pump before replacing them onto the instrument.2. Bring the polymer to room temperature, swirl to dissolve any deposits. Replace polymer if expired.3. Perform new spectral calibration.4. Place a drop of methanol onto the detection cell window.

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Observation	Possible Cause	Recommended Action
Loss of resolution.	<ol style="list-style-type: none"> 1. Too much sample injected. 2. Poor quality water. 3. Poor quality or dilute running buffer. 4. Poor quality or breakdown of polymer. 5. Capillary array used for more than 150 injections. 6. Degraded formamide. 7. High salt concentration. 8. Improper injection and run conditions 	<ol style="list-style-type: none"> 1. Dilute the sample and reinject. 2. Use high quality, ultra pure water. 3. Prepare fresh running buffer. 4. Use a fresh lot of polymer. 5. Replace with new capillary array. 6. Use fresh formamide and ensure correct storage conditions. 7. Use a recommended protocol (e.g., microcon) for salt removal. Dilute salts with water. 8. Notify QA to check default settings.
Poor resolution in some capillaries.	Insufficient filling of array.	Refill array and look for cracked or broken capillaries. If problems persists contact Technical Support.
No current	<ol style="list-style-type: none"> 1. Poor quality water. 2. Water placed in buffer reservoir position 1. 3. Not enough buffer in anode reservoir. 4. Buffer is too dilute. 5. Bubbles present in the polymer block and/or the capillary and /or peek tubing. 	<ol style="list-style-type: none"> 1. Use high quality, ultra pure water. 2. Replace with fresh running buffer. 3. Add buffer up to fill line. 4. Prepare new running buffer. 5. Pause run and inspect the instrument for bubbles. They may be hidden in the peek tubing.
Elevated current.	<ol style="list-style-type: none"> 1. Decomposed polymer 2. Incorrect buffer dilution 3. Arcing in the gel block 	<ol style="list-style-type: none"> 1. Open fresh lot of polymer and store at 4°C. 2. Prepare fresh 1X running buffer. 3. Check for moisture in and around the septa, the reservoirs, the oven, and the autosampler

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Observation	Possible Cause	Recommended Action
Fluctuating current	<ol style="list-style-type: none"> 1. Bubble in polymer block. 2. A slow leak may be present in the system. 3. Incorrect buffer concentration. 4. Not enough buffer in anode. 5. Clogged capillary. 6. Arcing 	<ol style="list-style-type: none"> 1. Pause the run, check the polymer path for bubbles, and remove them if present. 2. Check polymer blocks and syringes for leaks. Tighten all fittings. 3. Prepare fresh running buffer. 4. Add buffer up to the fill line. 5. Refill capillary array and check for clogs. 6. Check for moisture in and around the septa, the reservoirs, the oven, and the autosampler.
Poor performance of capillary array used for fewer than 150 runs	<ol style="list-style-type: none"> 1. Poor quality formamide 2. Incorrect buffer 3. Poor quality sample, possible cleanup needed 	<ol style="list-style-type: none"> 1. Prepare fresh formamide and reprep samples 2. Prepare new running buffer 3. Desalt samples using a recommended purification protocol (e.g., microcon).
Migration time becomes progressively slower.	<ol style="list-style-type: none"> 1. Leak in the system. 2. Improper filling of polymer block. 3. Expired polymer. 	<ol style="list-style-type: none"> 1. Tighten all ferrules, screws and check valves. Replace any faulty parts. 2. Check polymer pump force. If the force needs to be adjusted, make a service call. 3. If necessary, change the lot of polymer.
Migration time becomes progressively faster.	Water in syringe resulting in diluted polymer.	Clean the syringe, make sure it is completely dry OR replace syringe.
Peaks exhibit a shoulder effect in GeneScan application.	Sample renaturation.	Heat-denature the sample in good-quality formamide and immediately place on ice.
Purging of polymer from the polymer reserve syringe.	<ol style="list-style-type: none"> 1. Arcing in the anode gel block. 2. Bubbles in syringes. 	<ol style="list-style-type: none"> 1. Replace the lower polymer block. 2. Remove bubbles.

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Observation	Possible Cause	Recommended Action
Leaking polymer at the top of either syringe.	Insufficient seal around the tip of the syringe plunger.	Do not move the syringe plunger when it is dry. Make sure to wet the plunger before filling the syringe with polymer. Do not mix and match barrels and plungers.
Leaking polymer at the bottom of the polymer-reserve syringe.	Improper tightening of the array ferrule knob to the syringe and/or to the polymer block.	Ensure the array ferrule knob is tightened.
Error message, "Leak detected" appears. The run aborts	1. Air bubbles in the polymer path. 2. Pump block system is loose/leaking. 3. Lower pump block has burnt out. When there is condensation in the reservoir(s) this will cause electrophoresis problems and burn the lower block	1. Check for bubbles and remove if present, then check for leaks. 2. Make sure all syringes, screws tubing is tightly secure. Ferrule in capillary end of block may be positioned wrong or missing. Check for this ferrule. 3. Replace the lower block.
Buffer jar fills very quickly with polymer.	1. Air bubbles in the polymer path. 2. Lower polymer block is not correctly mounted on the pin valve.	1. Check for bubbles and remove if present. Then, look for leaks. 2. Check to make sure the metal fork is in between the pin holder and not on top or below it.
Detection window pops out while replacing the capillary array. Replacing the window in the correct orientation is difficult.	Tightening of the array ferrule knob at the gel block causes high tension.	Loosen the array ferrule knob to allow the secure placement of the window. Retighten and close the detection door.

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Observation	Possible Cause	Recommended Action
Detection window stuck. It is difficult to remove when changing the capillary array.		To loosen the detection window: a. Undo the array ferrule knob and pull the polymer block towards you to first notch. b. Remove the capillary comb from the holder in the oven. c. Hold both sides of the capillary array around the detection window area, and apply gentle pressure equally on both sides. d. Release.

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For a run on the 3100 Genetic Analyzer, it is possible to run multiple sets of amplifications in one tray. **IMPORTANT:** Samples must be analyzed together with the controls from the same amplification sheet. Care has to be taken that the correct controls stay with each sample set.

A. YM1 GENOTYPER SECTION (3100)

To begin the YM1 Genotyper Software, double-click on the **YM1** icon.

1. Under **File** go to **Import** and select **From Genescan File**. This opens up the Add Sample Files window. Double-click on the folder containing the project that you created in Genescan.
2. To add the sample files, double-click on the project generated in the run. Click **Add** or double-click on the project icon to add the project for analysis.
3. When the project has been added, click **Finish**.
4. After importing the project, save the Genotyper as your initials and the run file name, e.g. STARS04-Y-001 CMK or STRIPES04-Y-001 JLS.
5. Run the Macro by pressing the **Control Key** and the **number 1** simultaneously, or by double-clicking on **POWER** in the lower left hand corner window. The plot window will appear automatically when the macro is completed. It will display orange size standard and all the peaks will be labeled with size in base pairs and allele names.

****Necessary GS 500 Standard Peaks: 9 Fragments from 139 to 400bp**

6. Check the results for the positive control.

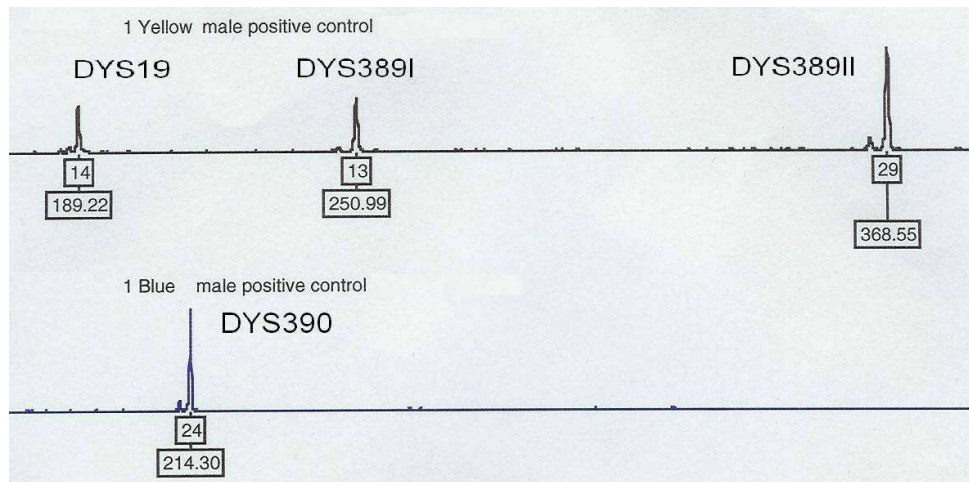
The genotype of the positive control is:

	DYS19	DYS389 I	DYS389 II
Yellow label	14	13	29
	DYS390		
Blue label	24		

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7. Check all lanes. Labels for extra peaks can be manually deleted by placing the cursor on the peak above the baseline and clicking.

Shortcut: If you mistakenly delete a label, before you do anything else, press **ctrl key + Z** and the allele name label will reappear (the command ctrl key + Z only undoes the last action).

To determine the size in bp for the editing documentation, click on the peak to remove the allele label. Click again to re-label with size in bp and then click again to finally remove the label. Use the zoom functions to get a close look at certain peaks (for instance if you have an allele with two labels which are very close to each other) by using the **Zoom** submenu under the **Views** menu.

Holding the left mouse click down draw a box around the desired area. Under **View** go to **Zoom**, select **Zoom In (selected area)**.

Shortcut: Zoom in by holding down the left mouse click button and dragging the cursor across the area you want to zoom in on. Then, simultaneously press the **Ctrl** and **R** keys to zoom in on that region.

To revert to the correct scan range, go to **View, Zoom**, and choose **Zoom To**. Set the plot range to **120 to 410**. Click **OK**.

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Peaks can be edited out if they meet one of the following criteria (also see section STR results interpretation):

- 1. Pull ups of peaks in any color caused by a very high peak of another color in the same lane. Pull ups are caused by the inability of the matrix file to remove all of the overlap.*
- 2. Shoulder peaks approximately 1-4 bp smaller or larger than the main allele. Shoulder peaks are mostly present on the right side of a peak if the peak shape shows a slope that is trailing out.*
- 3. "N" bands where the main allele shows a split peak. "N" bands are caused by incomplete extra A addition and are characterized by either a jagged edge on the left side of the peak or a complete split on the top level.*
- 4. -4 and +4 stutter peaks if there is no indication of the presence of a mixture. A -4 bp stutter is common and will often be labeled if followed by a high allele peak. A +4 bp stutter on the other side is extremely rare and has to be interpreted carefully. Defined peaks in a +4 bp position might indicate the presence of a mixture.*
- 5. Non specific artifacts. This category should be used if a labeled peak is caused by a not previously categorized technical problem.*
- 6. Labels placed on elevated or noisy baselines which do not resemble distinct peaks.*
- 7. Sharp peaks or spikes that do not resemble peaks but rather vertical lines and are caused by air bubbles or polymer crystals passing through the laser window. The presence of a spike can be verified by looking at the orange size standard in the same lane. A orange "spike" should occur at the same position.*
- 8. Dye artifact occurring at a constant scan range.*
8. Complete the Genotyper Editing Sheet for each sample number and note the reason for removal of a peak using the number code above.

At this stage it is also necessary to make decisions about samples that should be rerun with either more or less amount of amplification product.

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Samples without an orange size standard:

The lack of orange size standard will be first noted during Genescan analysis and can be confirmed on the Genotyper level. For 3100 runs these samples can be spotted easily since the Genotyper print out will state: "No Data Available." The lack of orange size standard can be either a failed injection or a post amplification sample prep mistake. These samples **MUST** be rerun under regular conditions.

Inconclusive samples:

Over-amplified samples often have peak heights between 5000 and 7000 fu's and are characterized by a plateau shaped or misshaped peaks and often contain a lot of labeled stutter peaks and artifacts (also see Interpretation of Complex Autosomal STR Results). Instead of laboriously editing out all of these peaks, the sample should be deemed inconclusive and marked for re-running. **All DNA mixtures where peaks in at least one color are ≥ 6000 fu's must be rerun with less.** Remove all labels from the lane in question, don't list all of the sizes, note "numerous" for peaks removed. The sample should be placed on a rerun sheet for rerunning with 1 μ L of a 1/10 dilution of the amplification product.

DNA alleles visible but below threshold:

If a sample displays allele peaks just below the instrument detection threshold there is a distinct possibility that the alleles can be identified after a repeated run with increased amplification product. Place the sample on a rerun sheet. **Use 4 μ L of amplified sample with the Rerun Module (10sec injection / 5KV).** Refer to Section E5 for appropriate letters for the system on a sample sheet.

New alleles:

If a locus displays a distinct unlabeled peak this could be a "new" allele that is outside the defined allele range. Especially if the other loci show a proper amplification signal, this possibility must be considered (but also see Troubleshooting F.). The presence of a possible "new" allele must be pointed out to a DNA supervisor for confirmation. The "new" allele will not be automatically reported in the Genotyper table, but will be visible on the electropherogram. Click on the unlabeled peak in order to label it with the size in bp, which is necessary for allele identification and reporting.

A "new" allele that does not match an exemplar must be rerun to eliminate the possibility of an electrophoresis shift.

9. After the editing has been finished scroll through the plot window to double check.

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10. Create a table by running the **Table Macro**.

Simultaneously press **Ctrl** key and the **number 2**, table will open.

Compare the sample information in the table with the amplification and the run control sheet. If an error gets detected at this point it can be corrected as follows:

- Open the dye/lane window or “sample info box”
- Place the cursor in the sample info box and correct the text
- Clear the table by going to **Analysis** on the main menu, select **Clear Table**
- Select the appropriate colors by shift clicking on the dye buttons or using edit
- Run **Table Macro** again

Continue to Step 12 and print the controls according to the directions. After the printing has finished, continue with Step 11.

11. Peak height labels are to be added to evidence-sample runs only. When processing, select the controls and case samples separately in order to add peak height labels to the evidence sample electropherograms. Follow the direction below for the different types of samples:

EVIDENCE SAMPLE RUNS

- a. Open the **Dye Lane Window** (under **View**) and select **blue and yellow** for all lanes containing controls (allelic ladder, positive control, and all negative controls). In order to select multiple labels it is necessary to hold down the Ctrl-key while clicking on the lanes. The controls might not all be at the beginning of the run, do not forget any microcon samples placed elsewhere. Go to **View** and open the **Plot Window**.

Under **Analysis** select **Change Labels**. A dialogue box appears giving you the option of labels to assign to the peaks. Select **Size in bp** and **Category name**. Click **ok**. Continue to Step 12 and print the controls according to the directions. After the printing has finished, continue with b).

- b. After the printing has finished, open the **Dye Lane Window** again (under **View**) and select **blue and yellow** for all lanes containing casework-samples. In order to select multiple labels it is necessary to hold down the Ctrl-key while clicking on the lanes. For a large sample sheet, it may be easier to select all samples and de-select the controls. Go to **View** and open the **Plot Window**.

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Under **Analysis** select **Change Labels**. A dialogue box appears giving you the option of labels to assign to the peaks. Select **Size in bp**, **Peak Height**, and **Category name**. Click **ok**. Continue to Step 12 and print the samples according to the directions.

EXEMPLAR RUNS

Under **Analysis** select **Change Labels**. A dialogue box appears giving you the option of labels to assign to the peaks. Select **Size in bp** and **Category name**. Click **ok**. Continue to Step 12 and print according to the directions.

12. Before printing the results make sure the file is named properly, including initials. Set Plot window zoom range as shown below. The active window will be printed so open Table and Plot as needed.
13. The Genotyper printout for YM1 should have a standard format: yellow lanes, then blue lanes. The table should have 2 rows for each locus.

Default settings are: Dye lane sorting: 1. Lane number - ascending
2. Dye color - descending

14. WINDOWS: Go to **File > Print**. Click **OK**. Click **Properties**. Select orientation. Click **More**. Change scaling as indicated below. Click OK, OK, OK.

YM1 Print out parameters:

	Table	Plot
Orientation	Portrait	Portrait
Scale	100% 2 per page	100%
Zoom range	n/a	120 - 410

15. After the printing is finished, under **file**, **quit** Genotyper. Click **save**.

Normally the software will place the Genotyper file to the folder from which the data were imported. Make sure that the Genotyper is saved in the appropriate **Common runs folder**.

16. Archive data as described in the archiving section.

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17. Initial all Genotyper pages. Pull the rerun samples. Have a Criminalist IV supervisor review the analyzed gel and get a signature on the editing sheet.

The Criminalist IV supervisor must go back to the GenoTyper file to review the orange size standard. This review shall be documented on the Editing Sheet.

B. COFILER AND PROFILER PLUS GENOTYPER SECTION

For 310 and 3100 instruments, it is possible to run multiple sets of amplifications in one tray. If the amplifications were done in different multiplex systems it is necessary to perform the Genotyper analysis separately using the appropriate templates. For two amplifications in the same system it is optional to process them together or separately.

IMPORTANT: Samples must be analyzed together with the controls from the same amplification sheet. Care has to be taken that the correct controls stay with each sample set.

1. Under **File** go to **Import** and select **From GeneScan File**. If the Current Runs folder does not already appear in the window, scroll to find it from the pull-down menu and double-click on it. Double-click on the folder containing the project that you created in Genescan.

MAC: Highlight project file, click **Import**.

WINDOWS: Double-click on the project generated in the run. Click **Add** or double-click on the project icon to add the project for analysis. When the project has been added, click **Finish**.

Under **View** select **Show Dye/Lanes window** you will see a list of the samples you have imported from Genescan analysis. If samples need to be removed, highlight the lanes for these samples and select **Cut** from the **Edit** menu.

2. Change the name of the Genotyper template to your initials and the casework run file name (under **File** select **Save As**).

e.g. Stripes04-Co001 LAR, Stars05-Pro001 EL

Or

CE3/04-005 Co LMS, CE1/05-002 Pro LAD

3. After importing the project and saving the Genotyper file run the first Macro.

MAC: simultaneously press **Apple key** and the **number 1**, or double click "**kazam**"

WINDOWS: simultaneously press **Ctrl key** and the **number 1**, or double click "**kazam**"

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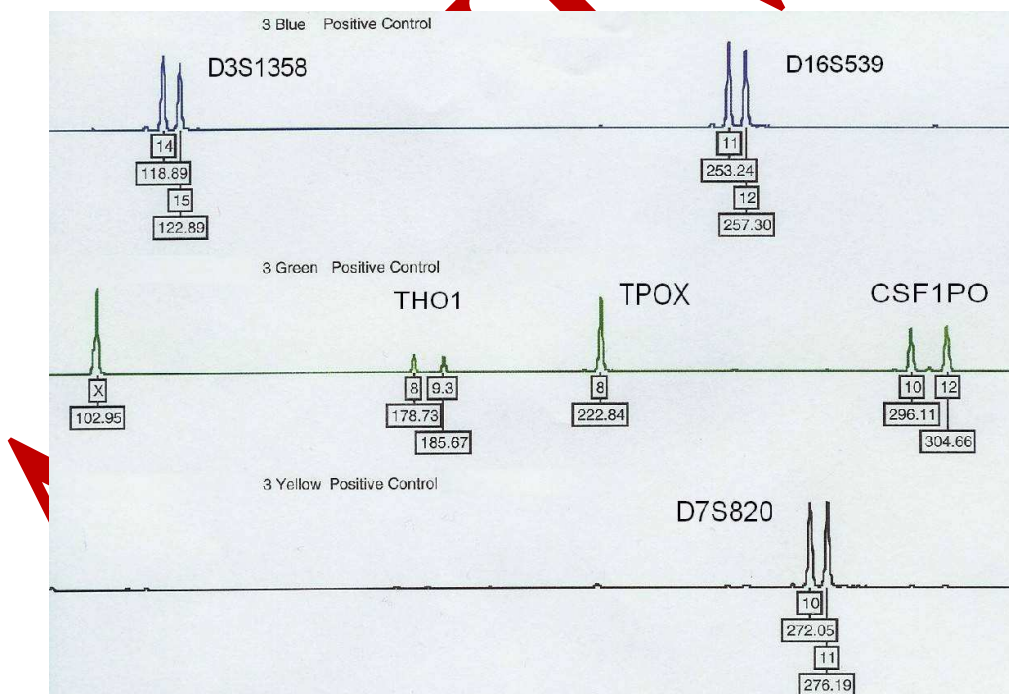
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4. The plot window will appear automatically when the macro is completed. Check to make sure that the ladders that were run match the allele sequence shown below. Also check the results for the positive control.

Multiplex System	Necessary GS500 standard peaks
Cofiler	10 fragments from 75 - 350 bp
Profiler Plus	11 fragments from 75 - 400 bp

The genotype of the Cofiler Positive Control is:

	D3S1358	D16S539		
Blue Label	14, 15	11, 12		
	Amelogenin	THO1	TPOX	CSF1PO
Green Label	X	8, 9, 9.3	8	10, 12
	D7S820			
Yellow Label	10, 11			



(Basepair sizes in the figure above correspond to an ABI 3100 capillary run.)

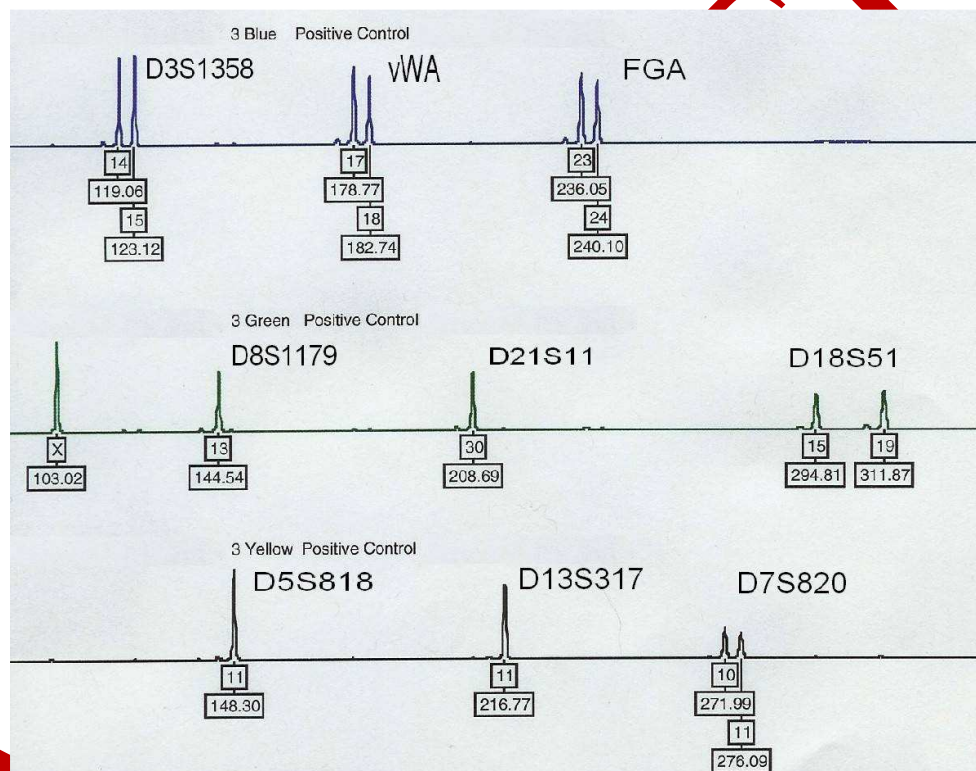
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The genotype of the Profiler Plus Positive Control is:

	D3S1358	VWA	FGA	
Blue Label	14, 15	17, 18	23, 24	
	Amelogenin	D8S1179	D21S11	D18S818
Green Label	X	13	30	15, 19
	D5S818	D13S317	D7S820	
Yellow Label	11	11	10, 11	



(Basepair sizes in the figure above correspond to an ABI 3100 capillary run.)

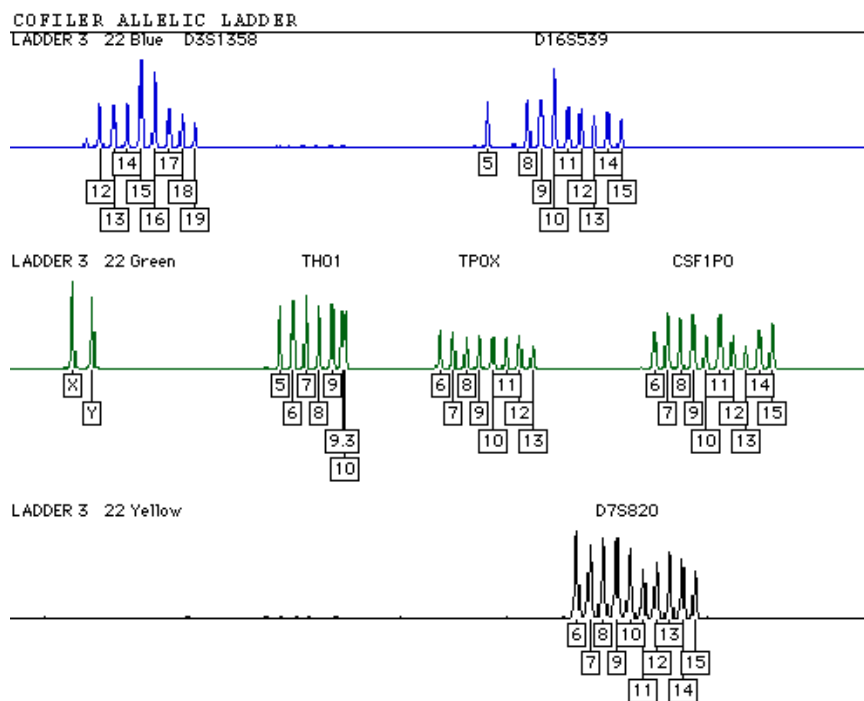
If the alleles for the positive control are shifted one step towards a higher allele number, this is an indication that the first allele of the allelic ladder has been assigned incorrectly (see troubleshooting).

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Cofiler Allelic Ladder:

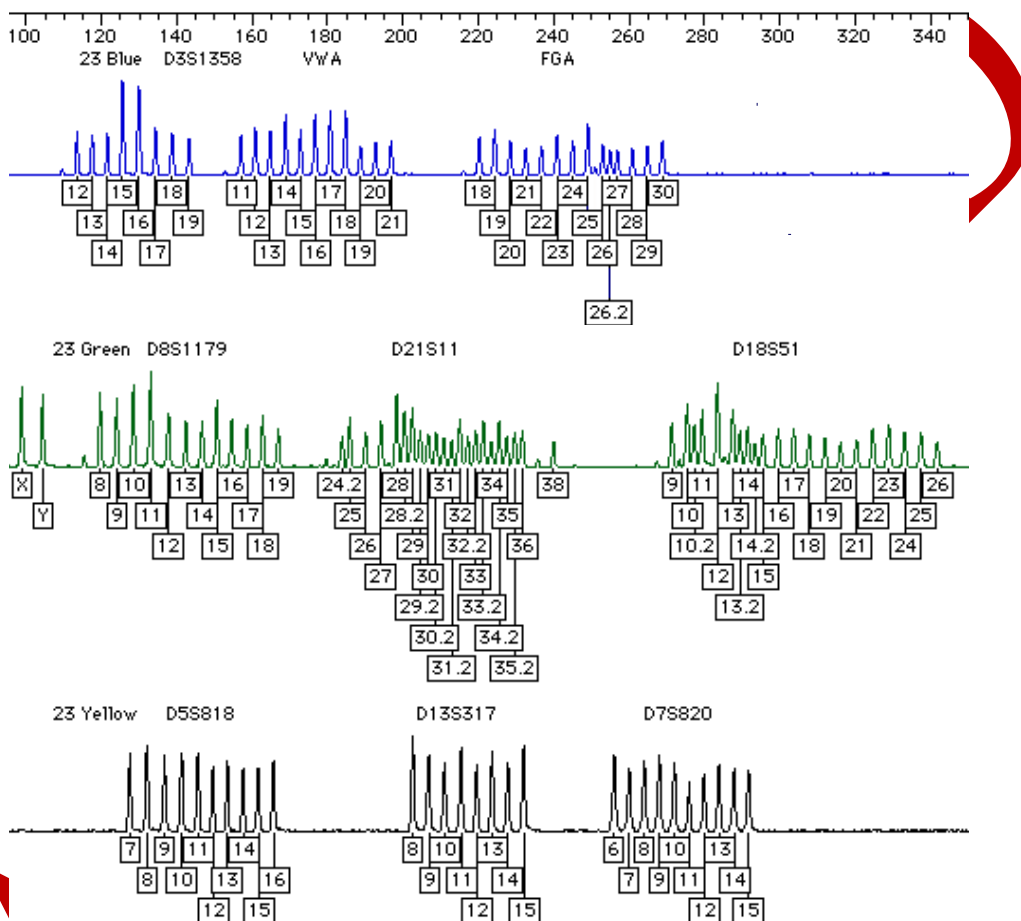


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Profiler Plus Allelic Ladder



5.

Check all lanes. Labels for extra peaks can be manually deleted by placing the cursor on the peak above the baseline and clicking.

Shortcut: If you mistakenly delete a label, before you do anything else, press the **apple (or ctrl) key + Z** and the allele name label will reappear (the command apple or ctrl key +Z only undoes the last action).

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To determine the size in bp for the editing documentation, click on the peak to remove the allele label. Click again to re-label with size in bp and then click again to finally remove the label. Use the zoom functions to get a close look at certain peaks (for instance if you have an allele with two labels which are very close to each other) by using the **Zoom** submenu under the **Views** menu.

Holding the left mouse click down draw a box around the desired area. Under **View** go to **Zoom**, select **Zoom In (selected area)**.

Shortcut: Zoom in by holding down the left mouse click button and dragging the cursor across the area you want to zoom in on. Then, simultaneously press the **apple (or Ctrl)** and **R** keys to zoom in on that region.

To revert to the correct scan range, go to **View, Zoom**, and choose **Zoom To**. Set the plot range to **90 to 330**. Click **OK**.

Peaks can be edited out if they meet one of the following criteria (also see section STR results interpretation):

1. *Pull-ups of peaks in any color caused by a very high peak of another color in the same lane. Pull-ups are caused by the inability of the matrix file to remove all overlap, e.g. green light components from a blue signal.*
2. *Shoulder peaks approximately 1-4 bp smaller or bigger than the main allele. Shoulder peaks are mostly present on the right side of a peak if the peak shape shows a slope that is trailing out.*
3. *"N" bands where the main allele shows a split peak. "N" bands are caused by incomplete extra A addition and are characterized by either a jagged edge on the left side of the peak or a complete split on the top level.*
4. *-4 and +4 bp stutter peaks if there is no indication of the presence of a mixture. -4 bp stutter is common and will often be labeled if followed by a high allele peak. +4 bp stutter on the other side is extremely rare and has to be interpreted carefully. Defined peaks in a +4 bp position might indicate the presence of a mixture.*
5. *Non-specific artifacts. This category should be used if a labeled peak is caused by a not previously categorized technical problem.*

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6. *Labels placed on elevated or noisy baselines, which do not resemble distinct peaks.*
7. *Sharp peaks (spikes) that do not resemble peaks but rather vertical lines and are caused by air bubbles or POP4 crystals passing the laser window. The presence of a spike can be verified by looking at the red size standard for the same lane (see below). A red “spike” should occur at the same position.*
8. *Dye artifact peak, which is defined as a “blip”, whose shape is not that of a regular peak, occurs at a constant scan position approximately between 3500 and 4000 and most frequently for the blue label. This peak is distinguishable from a normal allele by its indeterminate shape. If you are uncertain, use the zoom options in order to view the peak(s) more closely.*

To compare the red electropherogram with the other color lanes, hold down the shift key and either click on the red “R” box in the upper left hand corner, or under **edit** go to **select** +red. Before printing the plots the red electropherograms must be deselected, and the other three colors re-selected as above.

6. For all instrument platforms, fill out the Genotyper Editing Sheet for each sample number and note the reason for removal of a peak using the number code above.

At this stage it is also necessary to make decisions about samples that should be rerun with either more or less amount of amplification product.

Red size standard:

For the 310, the lack of red size standard will be noted during Genescan analysis and can be confirmed on the Genotyper level. For the 3100 runs, these samples can be spotted easily since the Genotyper print out will state: “No Data Available.” The lack of red size standard can be either a failed injection or a post-amplification sample prep mistake. These samples **MUST** be rerun under regular conditions.

Samples with compromised red size standards must either be re-analyzed with a re-defined size standard or designated for rerun.

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Inconclusive samples:

Over-amplified samples often have peak heights between 5000 and 7000 fu's and are characterized by a plateau shape or misshaped peaks, and often contain a lot of labeled stutter peaks and artifacts (also see Interpretation of Complex Autosomal STR Results). Instead of laboriously editing out all of these peaks, the sample should be deemed inconclusive and marked for re-running. **All DNA mixtures where peaks in at least one color are ≥ 6000 fu's have to be rerun with less amp product.** Remove all labels from the lane in question, don't list all of the sizes, note "numerous" for peaks removed. The sample should be placed on a rerun sheet for rerunning with 1 μ L of a 1/10 dilution of the amplification product.

DNA alleles visible but below threshold:

If a sample displays allele peaks just below the instrument detection threshold there is a distinct possibility that the alleles can be identified after a repeated run with either increased amplification product or a prolonged injection time. Place the sample on a rerun sheet.

310: use 2 μ L amplified sample and 10 seconds injection

3100: use 4 μ L of amplified product with the Rerun Module (20 second injection/5 KVolts)

New alleles:

If a locus displays only one peak label and a distinct same color peak is visible but is not labeled, or is labeled "OL allele?" it is because the unlabeled peak is outside the defined allele range or is not present in the allelic ladder. This peak might be a "new", previously unreported allele. This possibility must be considered, especially if the other loci show a proper allele profile. The "new" allele will not be automatically reported in the Genotyper table, but will be visible on the electropherogram. Click on the unlabeled peak in order to label it with the size in bp, which is necessary for allele identification.

NOTE: OL alleles that were not duplicated in an overlapping system and do not match an exemplar or another sample in the case must be rerun in order to exclude the possibility of an electrophoresis shift.

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7. After the editing has been finished scroll through the plot window to double-check.

8. Create a table by running the **Create Table Macro**.

MAC: simultaneously press **Apple key** and the **number 2**, under **View** open the table.

WINDOWS: simultaneously press **Ctrl key** and the **number 2**, table will open.

Compare the sample information in the table with the amplification and the run control sheet. If an error is detected at this point it can be corrected as follows:

- Open the dye/lane window or "sample info box"
- Place the cursor in the sample info box and correct the text
- Clear the table by going to **Analysis** on the main menu, select **Clear Table**
- Select the appropriate colors by shift clicking on the dye buttons or using edit
- Run **Create Table Macro** again

Continue to Step 10 and print the controls according to the directions. After the printing has finished, continue with Step 9.

9. Peak height labels are to be added to evidence-sample runs only. When processing, select the controls and case samples separately in order to add peak height labels to the evidence sample electropherograms. Follow the direction below for the different types of samples:

EVIDENCE SAMPLE RUNS

- Open the **Dye Lane Window** (under **View**) and select **blue, green, and yellow** for all lanes containing controls (allelic ladder, positive control, and all negative controls). In order to select multiple labels it is necessary to hold down the Ctrl-key while clicking on the lanes. The controls might not all be at the beginning of the run, do not forget any microcon samples placed elsewhere. Go to **View** and open the **Plot Window**.

Under **Analysis** select **Change Labels**. A dialogue box appears giving you the option of labels to assign to the peaks. Select **Size in bp** and **Category name**. Click **ok**. Continue to Step 10 and print the controls according to the directions. After the printing has finished, continue with b).

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- b. After the printing has finished, open the **Dye Lane Window** again (under **View**) and select **blue, green, and yellow** for all lanes containing casework-samples. In order to select multiple labels it is necessary to hold down the Ctrl-key while clicking on the lanes. For a large sample sheet, it may be easier to select all samples and de-select the controls. Go to **View** and open the **Plot Window**.

Under **Analysis** select **Change Labels**. A dialogue box appears giving you the option of labels to assign to the peaks. Select **Size in bp**, **Peak Height**, and **Category name**. Click **ok**. Continue to Step 10 and print the controls according to the directions.

EXEMPLAR RUNS

Under **Analysis** select **Change Labels**. A dialogue box appears giving you the option of labels to assign to the peaks. Select **Size in bp** and **Category name**. Click **ok**. Continue to Step 10 and print according to the directions.

10. Before printing the results make sure the file is named properly, including initials. Set Plot window zoom range as shown below. The active window will be printed so open Table and Plot as needed.
11. MAC: Under **file** change the **page set-up** to settings specified below. Then select **print**. WINDOWS: Go to **File > Print**. Click **OK**. Click **Properties**. Select orientation. Click **More**. Change scaling as indicated below. Click OK, OK, OK.

Cofiler print parameters:

	Table		Plot	
	MAC	WINDOWS	MAC	WINDOWS
Orientation	Landscape	Landscape	Letter	Portrait
Scale	65%	70%	90%	90%
Zoom range	n/a	n/a	90 - 330	90 - 330

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Profiler Plus Print out parameters

	Table		Plot	
	MAC	WINDOWS	MAC	WINDOWS
Orientation	Landscape	Landscape	Letter	Portrait
Scale	65%	70%	90%	90%
Zoom range	n/a	n/a	90 - 360	90 - 360

12. After the printing is finished, under **file, quit** Genotyper. Click **save**.

MAC: The Genotyper file will automatically be saved in the run folder from which you imported your data; it can be located there and re-edited at a later date.

WINDOWS: Normally the software will place the Genotyper file to the folder from which the data were imported. Make sure that the Genotyper is saved in the appropriate **Common runs folder**.

13. Archive data as described in the archiving section.
14. Initial all Genotyper pages. Pull the rerun samples and list on the appropriate rerun sheet. Have a supervisor review the analyzed gel and get a signature on the editing sheet.

For **Troubleshooting** see Genotyper section **E Multiplex Kit Troubleshooting**.

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C. POWERPLEX 16 GENOTYPER SECTION

For a run on the 3100 Genetic Analyzer, it is possible to run 85 samples (including PE and amp. negative). **IMPORTANT:** Samples must be analyzed together with the controls from the same amplification sheet. Care has to be taken that the correct controls stay with each sample set.

To begin the PowerPlex 16 Genotyper Software, double-click on the **PowerPlex 16** icon.

1. Under **File** go to **Import** and select **From Genescan File**. This opens up the Add Sample Files window. Double-click on the folder containing the project that you created in Genescan.
2. To add the sample files, double-click on the project generated in the run. Click **Add** or double-click on the project icon to add the project for analysis.
3. When the project has been added, click **Finish**.
4. After importing the project, save the genotyper as your initials and the run file name, e.g. STARS04-PP16-001 CMK or STRIPES04-PP16-001 JLS.
5. Run the Macro by pressing the **Control Key** and the **number 1** simultaneously, or by double-clicking on **POWER** in the lower left hand corner window. The plot window will appear automatically when the macro is completed.
6. Check the allelic ladder (paying close attention to FGA alleles 18.2, 19.2, 20.2, 21.2, 22.2) and click on the 0.2 peaks that were not labeled by the macro. (The ladder has worked correctly but a default filter has removed those labels. You are just replacing those labels).
7. Go to **Change Labels** from **Analysis** in the main menu and check the boxes for the **Category Name** and the **Base Pair** values. Click **OK**.

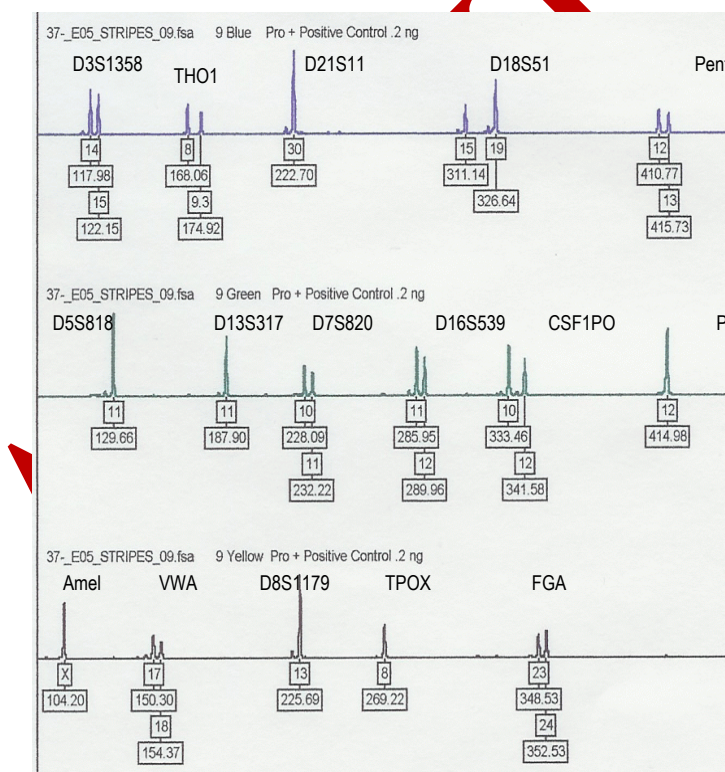
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8. Check the results for the positive control.

The genotype of the PowerPlex 16 Positive Control is:

	D3S1358	THO1	D21S11	D18S51	Penta E	
Blue Label	14, 15	8, 9.3	30	15, 19	12, 13	
	D5S818	D13S317	D7S820	D16S539	CSF1PO	Penta D
Green Label	11	11	10, 11	11, 12	10, 12	12
	vWA	D8S1179	TPOX	FGA		
Yellow Label	17, 18	13	8	23, 24		



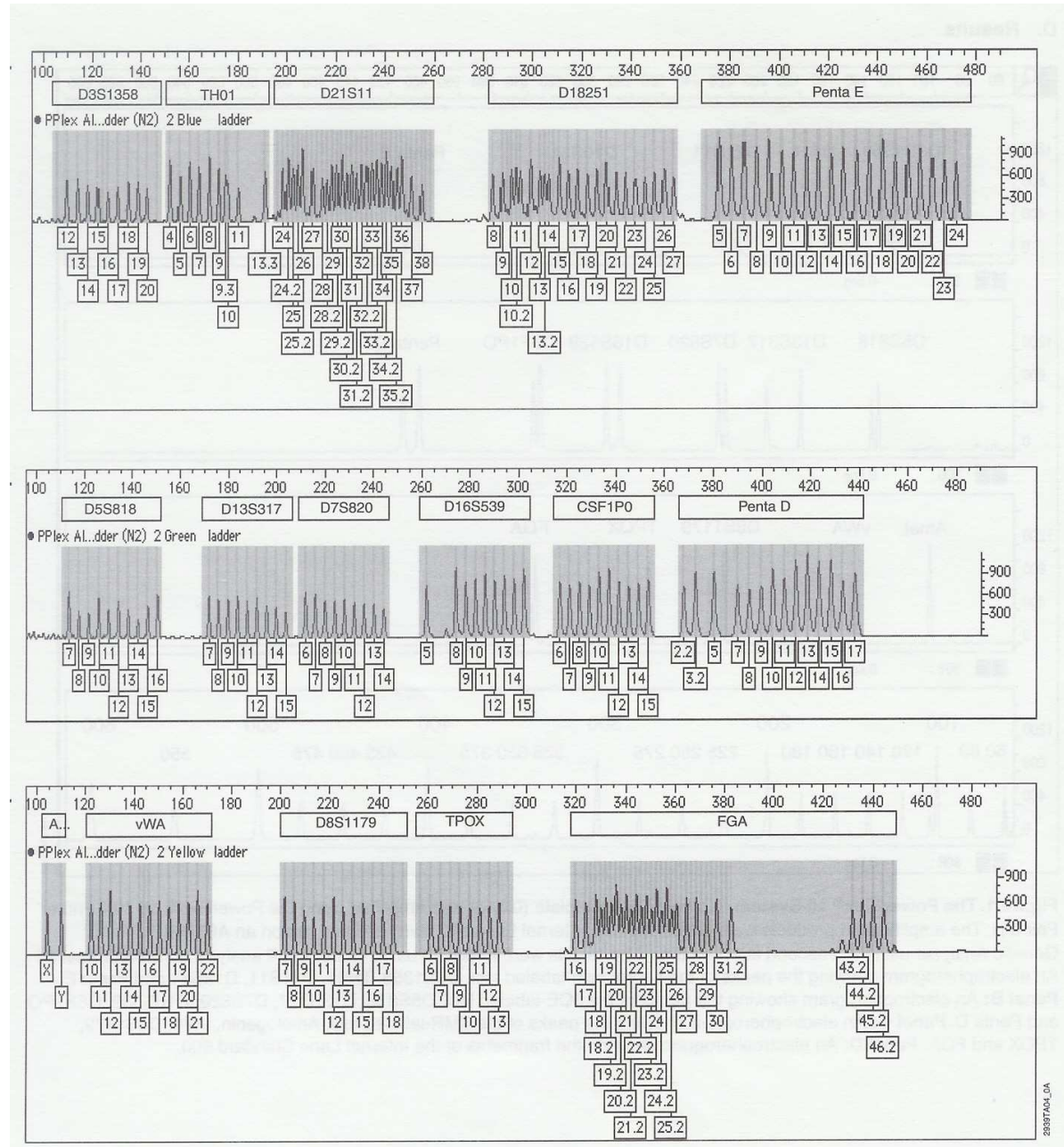
If the alleles for the positive control are shifted one step towards a higher allele number, this is an indication that the first allele for the allelic ladder has been assigned incorrectly (see troubleshooting section).

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PowerPlex 16 Allelic Ladder



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9. Check all lanes. Labels for extra peaks can be manually deleted by placing the cursor on the peak above the baseline and clicking.

Shortcut: If you mistakenly delete a label, before you do anything else, press the **apple (or ctrl) key + Z** and the allele name label will reappear (the command apple or ctrl key +Z only undoes the last action).

To determine the size in bp for the editing documentation, click on the peak to remove the allele label. Click again to re-label with size in bp and then click again to finally remove the label. Use the zoom functions to get a close look at certain peaks (for instance if you have an allele with two labels which are very close to each other) by using the **Zoom** submenu under the **Views** menu.

Holding the left mouse click down draw a box around the desired area. Under **View** go to **Zoom**, select **Zoom In (selected area)**.

Shortcut: Zoom in by holding down the left mouse click button and dragging the cursor across the area you want to zoom in on. Then, simultaneously press the **apple (or Ctrl)** and **R** keys to zoom in on that region.

To revert to the correct scan range go to **View, Zoom**, and choose **Zoom To**. Set the plot range to **90 to 480**. Click **OK**.

Peaks can be edited out if they meet one of the following criteria (also see section STR results interpretation):

1. *Pull ups of peaks in any color caused by a very high peak of another color in the same lane. Pull ups are caused by the inability of the matrix file to remove all of the overlap.*
2. *Shoulder peaks approximately 1-4 bp smaller or larger than the main allele. Shoulder peaks are mostly present on the right side of a peak if the peak shape shows a slope that is trailing out.*
3. *“N” bands where the main allele shows a split peak. “N” bands are caused by incomplete extra A addition and are characterized by either a jagged edge on the left side of the peak or a complete split on the top level.*

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4. *-4 and +4 stutter peaks if there is no indication of the presence of a mixture. A -4 bp stutter is common and will often be labeled if followed by a high allele peak. A +4 bp stutter on the other side is extremely rare and has to be interpreted carefully. Defined peaks in a +4 bp position might indicate the presence of a mixture.*
5. *Non specific artifacts. This category should be used if a labeled peak is caused by a not previously categorized technical problem. (This category should be chosen for the artifacts prevalent in vWA.)*
6. *Labels placed on elevated or noisy baselines which do not resemble distinct peaks.*
7. *Sharp peaks or spikes that do not resemble peaks but rather vertical lines and are caused by air bubbles or polymer crystals passing through the laser window. The presence of a spike can be verified by looking at the red size standard in the same lane. A red "spike" should occur at the same position.*
8. *Dye artifact occurring at a constant scan range.*

To compare the red electropherogram with the other color lanes, hold down the shift key and either click on the red "R" box in the upper left hand corner, or under **edit** go to **select +red**. Before printing the plots the red electropherograms must be deselected, and the other three colors re-selected as above.

10. Fill out the Genotyper Editing Sheet for each sample number and note the reason for removal of a peak using the number code above.

At this stage it is also necessary to make decisions about samples that should be rerun with either more or less amount of amplification product.

Samples without a red size standard:

The lack of red size standard will be first noted during Genescan analysis and can be confirmed on the Genotyper level. For the 3100 runs these samples can be spotted easily since the Genotyper print out will state: "No Data Available." The lack of red size standard can be either a failed injection or a post-amplification sample prep mistake. These samples **MUST** be rerun under regular conditions.

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Inconclusive samples:

Over-amplified samples often have peak heights between 5000 and 7000 fu's and are characterized by a plateau shape or misshaped peaks and often contain a lot of labeled stutter peaks and artifacts (also see Interpretation of Complex Autosomal STR Results). Instead of laboriously editing out all of these peaks, the sample should be deemed inconclusive and marked for re-running. **All DNA mixtures where peaks in at least one color are ≥ 6000 fu's have to be rerun with less.** Remove all labels from the lane in question, don't list all of the sizes, note "numerous" for peaks removed. The sample should be placed on a rerun sheet for rerunning with 1 μ L of a 1/10 dilution of the amplification product.

DNA alleles visible but below threshold:

If a sample displays allele peaks just below the instrument detection threshold there is a distinct possibility that the alleles can be identified after a repeated run with increased amplification product. Place the sample on a rerun sheet.

3100: use 4 μ L of amplified product with the Rerun Module (20 second injection/5 KVolts)

New alleles:

If a locus displays only one peak label and a distinct same color peak is visible but is not labeled, or is labeled "OL allele?", it is because the unlabeled peak is outside the defined allele range or is not present in the allelic ladder. This peak might be a "new", previously unreported allele. This possibility must be considered, especially if the other loci show a proper allele profile. The "new" allele will not be automatically reported in the Genotyper table, but will be visible on the electropherogram. Click on the unlabeled peak in order to label it with the size in bp, which is necessary for allele identification.

NOTE: OL alleles that were not duplicated in an overlapping system and do not match an exemplar must be rerun in order to exclude the possibility of an electrophoresis shift.

11. After the editing has been finished scroll through the plot window to double-check.
12. Create a table by running the **Create Table Macro**.

Simultaneously press **Ctrl** key and the **number 2**, table will open.

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Compare the sample information in the table with the amplification and the run control sheet. If an error gets detected at this point it can be corrected as follows:

- Open the dye/lane window or “sample info box”
- Place the cursor in the sample info box and correct the text
- Clear the table by going to **Analysis** on the main menu, select **Clear Table**
- Select the appropriate colors by shift clicking on the dye buttons or using edit
- Run **Create Table Macro** again

Continue to Step 14 and print the controls according to the directions. After the printing has finished, continue with Step 13.

13. Peak height labels are to be added to evidence-sample runs only. When processing, select the controls and case samples separately in order to add peak height labels to the evidence sample electropherograms. Follow the direction below for the different types of samples:

EVIDENCE SAMPLE RUNS

- a. Open the **Dye Lane Window** (under **View**) and select **blue, green, and yellow** for all lanes containing controls (allelic ladder, positive control, and all negative controls). In order to select multiple labels it is necessary to hold down the Ctrl-key while clicking on the lanes. The controls might not all be at the beginning of the run, do not forget any microcon samples placed elsewhere. Go to **View** and open the **Plot Window**.

Under **Analysis** select **Change Labels**. A dialogue box appears giving you the option of labels to assign to the peaks. Select **Size in bp** and **Category name**. Click **ok**. Continue to Step 14 and print the controls according to the directions. After the printing has finished, continue with b).

- b. After the printing has finished, open the **Dye Lane Window** again (under **View**) and select **blue, green, and yellow** for all lanes containing casework-samples. In order to select multiple labels it is necessary to hold down the Ctrl-key while clicking on the lanes. For a large sample sheet, it may be easier to select all samples and de-select the controls. Go to **View** and open the **Plot Window**.

Under **Analysis** select **Change Labels**. A dialogue box appears giving you the option of labels to assign to the peaks. Select **Size in bp**, **Peak Height**, and **Category name**. Click **ok**. Continue to Step 14 and print the controls according to the directions.

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EXEMPLAR RUNS

Under **Analysis** select **Change Labels**. A dialogue box appears giving you the option of labels to assign to the peaks. Select **Size in bp** and **Category name**. Click **ok**. Continue to Step 14 and print according to the directions.

14. Before printing the results make sure the file is named properly, including initials. Set Plot window zoom range as shown below. The active window will be printed so open Table and Plot as needed.
15. WINDOWS: Go to **File > Print**. Click **OK**. Click **Properties**. Select orientation. Click **More**. Change scaling as indicated below. Click OK, OK, OK.

Powerplex 16 Print out parameters

	Table	Plot
Orientation	Landscape	Portrait
Scale	70%	90%
Zoom range	n/a	90 - 480

16. After the printing is finished, under **file**, **quit** Genotyper. Click **save**.

Normally the software will place the Genotyper file in the folder from which the data were imported. Make sure that the Genotyper is saved in the appropriate **Common runs folder**.

17. Archive data as described in the archiving section.
18. Initial all Genotyper pages. Pull the rerun samples. Have a Criminalist IV supervisor review the analyzed gel and get a signature on the editing sheet.

The Criminalist IV supervisor must go back to the GenoTyper file to review the red size standard. This review shall be documented on the Editing Sheet.

For **Troubleshooting** see Genotyper section **E Multiplex Kit Troubleshooting**.

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D. Multiplex Kit Troubleshooting

1. If you get an Error Message when you try to run the Genotyper Macro 1 that reads:
“Could not complete your request because no dye/lanes are selected”.

Make sure you have actually imported the ladder from the project. Make sure “ladder” is spelled correctly in the **dye/lanes window**. If there is a misspelling or the sample information for the ladder is absent, the macro will not recognize the ladder and will not be able to complete the procedure. Correct the spelling and rerun the macro.

2. If you get an Error Message when you try to run the Genotyper Macro 1 that reads:
“Could not complete your request because the labeled peak could not be found”.

This message indicates that the ladder cannot be matched to the defined categories. There are four possibilities:

- a. The wrong ladder is being typed, (i.e. you are trying to type a Cofiler ladder in the Profiler Plus Genotyper or vice versa).

Solution: Close your Genotyper template, start the correct one and re-import your Genescan project.

- b. There may be peaks in the ladder that are too low to be recognized by the program.

Solution: You have two options:

One: If another ladder in the run is more intense, alter or delete the name of the first ladder in the Genotyper Dye/Lane window and rerun **Macro 1**. Now the macro will use the first backup ladder for the off-set calculation.

Two: You can lower the **minimum peak height** in the categories window. To do this, open the categories window by going under **Views** and selecting **Show Categories Window**. In the “offset” categories the first allele is defined with a scaled peak height of 200 or higher. The high value is meant to eliminate stutter and background.

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Change this to 75 for the 3100, and 100 for the 310, by clicking on the first category that highlights it. In the dialogue box locate the **Minimum Peak Height** and change it to the appropriate value, click **Add**, and then click **Replace** when given the option. You must do this for each locus. Do not use values less than the instrument threshold.

DO NOT CHANGE THE MINIMUM PEAK HEIGHT FOR ANY OTHER CATEGORY EXCEPT THE OFF-SET.

It is important, after you rerun the macro, to make sure the ladder begins with the correct allele and that the first allele is not assigned to a stutter which might precede the first peak.

- c. The first ladder peak of each locus is outside of the pre-defined size range window.

Solution: You can expand the search window in the categories window. To do this, open the categories window by going under **Views** and selecting **Show Categories Window**. In the “offset” categories the first allele is defined with a certain size +/- 7bp. Change the 7 to 10 or higher, by clicking on the first category which highlights it. In the dialogue box locate the +/- **box** and change the value, click **Add**, and then click **Replace** when given the option. You must do this for each locus that gives you the error message.

- d. There are no peaks at all in any of the allelic ladders.

Solution: Rerun all samples with freshly prepared Allelic Ladders.

2. Off Ladder (OL) allele labels

- a. If you have a run with a large number of samples you may find that the samples toward the end of the run have a high incidence of OL allele labels. This is due to a shift during the run.

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In order to improve the number of correctly called alleles, try to reanalyze the run by using the second allelic ladder as the off-set reference. This is done by removing the word “ladder” from the name of the first ladder in the dye lane window. This way this ladder is not recognized by the macro program. Rerun **Macro 1** and evaluate the results. Determine which one of both allelic ladders causes fewer “OL allele?” labels. Complete the Genotyping process using this ladder. Any remaining samples displaying OL alleles have to be rerun.

- a. If all or most of your samples have “OL allele?” labels it may be that your samples were automatically analyzed with an ill-defined size standard. In this case it is necessary to reanalyze your run using the correct or a redefined size standard (see Genescan analysis for 310 or 3100).
3. Incorrect positive control type:
 - a. The Genotyper has shifted allele positions during the category assignment to the ladder.

Check the ladder and make sure the first assigned allele is assigned to the first real peak and not to a stutter peak, which may precede it. If the stutter peak is designated with the first allele name, you must raise the peak height in the categories window in order to force the software to skip the stutter peak and start with proper allele.

- First determine the height of the stutter peak by placing the cursor on the peak in question (as if you are editing). The information displayed on the top of the window refers to the peak where the cursor is located and contains the peak height. Make a note of the peak height.
- Open the categories window (under views on the menu) and highlight the first allele in the offset category (e.g 18 o.s.) of the polymorphism that needs to be corrected.
- In the dialogue box change the height for the minimum peak height to a few points above the determined height of the stutter.
- Rerun the macro and then check to make sure everything is correct by looking at the first allele in each locus in the ladder and by comparing the result for the positive control

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- b. You have a sample mix-up and you have to rerun and/or to reamplify your samples.
4. Lining up unlabeled peaks

In order to place samples next to each other for comparison purposes, mark them by double clicking. A black bullet appears in front of the lane number. If this happens accidentally, a lane can be unmarked by either double clicking on it again or, under **Edit**, selecting **unmark**. NOTE: unsized peaks cannot be placed according to size on the electropherogram. Therefore, when comparing an unlabeled allele (unlabeled because it is too low to be sized, but high enough to be detected visually) to a labeled allele (e.g. in the ladder) you cannot determine the allele type and size by visual comparison while the results are viewed by size. To be able to align an unlabeled allele with a labeled allele in the same run you must select **View by Scan** from under the **View** menu!
5. Too many samples

If you see the same sample listed several times in the dye/lanes window or you see more samples than you have imported, you have most likely imported your samples more than once or you have imported your samples into a Genotyper template that already contained other samples. Under **Analysis** select **Clear Dye/Lanes** window and also under **Analysis** select **Clear Table**. Re-import your file(s).
6. Typographical error in the sample information and/or sample comment

If you detect a mistake in the sample information, this can be corrected for the Genotyper file by opening the dye lane list window, highlighting the lane, and retyping the sample information for all colors. The short sample name cannot be changed here. It can only be changed on the sample sheet level.
7. Less samples in Table than in Plots

Samples with the same sample information are only listed once in the Table. Add modifier to the sample information (see above) of one of the samples and rerun Macro.

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8. Too many background peaks labeled

If peaks are still labeled in the plot even though they are listed as having been removed or they appear to be below the stutter filter threshold, the following mistake could have happened: instead of choosing **Change labels** from the **Analysis** drop down menu, the analyst clicked **Label peaks**. The **Change labels** command is supposed to label the valid peaks with the allele name and the size in basepairs prior to printing the plot. The **Label peaks** command will label all peaks above threshold independent of any Macro stutter and background filters. This command will also re-label peaks that were edited out. To fix this, rerun the macro, repeat the documented editing steps and reprint the Table and the Plot.

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10. STR RESULTS INTERPRETATION

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A. Allele Calling Criteria

Results are interpreted by observing the occurrence of electropherogram peaks for the loci that are amplified simultaneously. The identification of a peak as an allele is determined through comparison to the allelic ladder or for YM1 by the Genotyper categories. An allele is characterized by the labeling color of the locus specific primers and the length of the amplified fragment. See appendix for a listing of each locus in each multiplex.

For each locus an individual can be either homozygous and show one allele, or heterozygous and show two alleles. In order to eliminate possible background and stutter peaks, only peaks that display intensity above the minimum threshold are labeled as alleles.

Based on validation data the different detection platforms have different minimum peak height thresholds:

ABI platform	Minimum threshold in fluorescent units
310	100 fu
3100	75 fu

The raw data collected by the Genescan Software undergo the following computer processing steps:

- Recalculating fluorescence peaks using the instrument-specific matrix or spectral file in order to correct for the overlapping spectra of the four fluorescent dyes.
- Calculating the fragment length for the detected peaks using the known in-lane standard fragments.
- For YM1 (a system without an allelic ladder) - labeling of all sized fragments that are >50 fu (fluorescent units), fall within the locus size range and match to an allele size average within a ± 1.0 bp tolerance window. Labels are automatically removed from minor peaks based on the background and stutter filter functions outlined in the YM1 Genotyper section.
- For Cofiler, Profiler Plus and Powerplex 16 (systems with an allelic ladder) - comparing and adjusting the Genotyper allele categories to the sizing of the co-electrophoresed allelic ladder by calculating the off sets (the difference between the first allele in a category and the first allele in the allelic ladder at each locus).

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- For Cofiler, Profiler Plus and Powerplex 16 - labeling of all sized fragments that are higher than the platform specific threshold and fall within the locus specific size range (see appendix). Removing the labels from minor peaks (background and stutter) according to the filter functions detailed in the appendix of this manual.

Additional **non-allelic peaks** may occur under the following instances (Clark 1988, Walsh et al. 1996, Clayton et al. 1998):

“Pull-ups” of peaks in one color caused by very high peaks in another color. This occurs only for multiplexes employing more than one labeling color and is caused by the inability of the software to compensate for the spectral overlap between the different colors, if the peak height is too high. Pull-ups are matrix artifacts.

Shoulder peaks approximately 1-4 bp smaller or larger than main allele (mostly larger), caused by a flat decline of main peak fluorescence. Shoulders are easily recognized because they do not have the shape of an actual peak.

-4 stutter peaks that are caused by slippage of the Taq polymerase enzyme during copying of the STR allele. Under rare circumstances there may be a +4 stutter peak.

Non-specific artifacts caused by non-specific priming in a multiplex reaction. These artifacts are usually easily recognized due to their low peak height and their position outside of the allele range.

Labels placed on elevated or noisy baselines which do not resemble distinct peaks. Noisy and elevated baselines are matrix artifacts.

“N” bands, where the main peak is split into two peaks caused by the Taq polymerase activity that causes the addition of a single A to the terminus of the amplified product (“N+1” band). The allele calling is based on the N+1 bands, therefore complete extra A addition is desired.

A constant peak at 3500-4000 scans. This peak is caused by fluorescent dye that is not attached to the primers anymore. These “color blips” can occur in all colors. The “color blip” falls into the allele range of D3S1358 and is therefore labeled if it occurs in blue.

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Sharp spikes caused by power surges or crystals or air bubbles traveling by the laser detector window. Spikes might look like a single vertical line or a peak; they can easily be distinguished from DNA peaks by looking at the other fluorescent colors, including red; the spike is usually present in all colors.

If the Genotyper program labels these additional non-allelic peaks, the labels may be removed manually. The removal is documented on the Genotyper editing sheet.

After the assigning of allele names to the remaining labeled peaks the Genotyper software prepares a result table where all peaks that meet the above listed criteria are listed as alleles. The allele nomenclature follows the recommendations of the International Society for Forensic Haemogenetics (ISFH), (DNA recommendations, 1994) and reflects the number of 4bp core repeat units for the different alleles. Subtypes displaying incomplete repeat units are labeled with the number of complete repeats and a period followed by the number of additional bases.

The Y chromosome allele nomenclature is also based on the number of 4bp core repeats and follows the nomenclature suggested in (Kayser et al 1997) and the one used in the European Caucasian Y-STR Haplotype database (Roewer et al 2001).

B. Interpretation of the Genotyper Print Out

Genotyper printouts of capillary electrophoresis runs containing the case specific samples are part of each casefile. The Genotyper results table reflects the number and allele assignments of the labeled peaks visible on the Genotyper Plot print out. **The printouts are the basis for results interpretation.** The plot will display peak height information, unlabeled peaks, intensity differences that indicate the presence of a mixture, and will show all peaks at one locus if the number of peaks exceeds the table's number of columns per locus. Looking at the plots also serves as a control for the editing process.

When to access the electronic Genotyper file:

1. No peak is above the minimum threshold but unlabeled peaks are visible.

If these unlabeled peaks need to be compared to a previous amplification (see reporting rules) it is important to remember that peaks below the threshold are not sized by the software and cannot be displayed by size. They are randomly placed on the Genotyper plot and their types cannot be inferred! In order to make a visual allele interpretation one must reopen the corresponding Genotyper file and change the viewing mode from **View by Size** to **View by Scan** (also see Genotyper troubleshooting section E).

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2. High peaks and very minor peaks present in the same color lane

Since the fu scale of the electropherogram is based on the highest peak in each color, alleles at weak loci will not be clearly visible if the loci are imbalanced. For mixture interpretation or allelic dropout detection, it is helpful to access the archived Genotyper file and in the **View** menu enter a fixed y-scale for **Plot Options, Main Window Lower Panel**. Print pages. Do not save changes.

When the Genotyper print outs are not sufficient for proper interpretation and the Genescan Analysis results can be helpful:

1. Genotyper plot states “no size data”

This means that none of the peaks was above threshold and it is possible that the original Genescan Analysis electropherogram displays visible peaks below the sizing threshold.

2. Distinct unlabeled peak in locus with similar height “homozygous” allele

If a sample has only one allele at a locus but outside the previously reported size range a distinct peak is visible, this sample could have a “new” allele (see Interpretation of complex STR results). In this case the analyst might have to look at the original Genescan Analysis electropherogram in order to get the size in bp for this peak.

C. Reporting Procedures

Items listed in allele typing tables should be limited to samples that are used to draw important conclusions of the case. Genotypes are not reported and should not be inferred, i.e. if only a “7” allele is found; it should be reported as 7. Alleles and/or peaks are listed in the results tables regardless of intensity differences, based on the reporting criteria below.

- A. If an allele meets the above reporting thresholds and fulfills the concordant analyses and the duplicate rule as stated in the General PCR Guidelines, then the allele is listed in the report.
- B. Use * to indicate that an extract was not tested for this locus. This symbol is defined as “* = Typing not attempted.”

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- C. If extra peaks are visible that can't be accounted for by any editing categories, then the presence of this peak is reported as **. ** is defined at the bottom of the table as “** = additional peaks were detected which did not meet laboratory criteria for allele identification; therefore, these additional peaks are not reported.”
- D. In cases where a mixture sample was reamplified in the same multiplex system consult the table below about how to report the alleles:

Allele labeled for one amplification?	Results for the other amplification:	Report
Yes	also labeled	allele
Yes	not labeled but visible	allele
Yes	not present at all	**
No but visible	not labeled but visible	**
No but visible	not present at all	**

NOTE: the table above does not apply to reanalysis by repeated electrophoresis or to single source samples that are amplified in varying DNA amounts.

- E. If no alleles are detected in a locus then the locus is reported as “NEG = no alleles detected.”
- F. Off-ladder alleles should be reported using the CODIS nomenclature which incorporates their relative position to the alleles in the allelic ladder (see Interpretation of Complex Autosomal STR results, 3.B. reporting of previously unreported rare alleles).
- G. New alleles observed for YM1 where no allelic ladder is available should be reported with their rounded basepair size. The basepair value should also appear in the footnotes, e.g. 128 = Allele is reported as size in basepairs.
- H. Other symbols or reporting procedures will be used if necessary depending on the details of the case.

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D. Comparison of Samples and Interpretation of Results in Report

- A. Determine whether it is likely that a sample contains a mixture of DNA (i.e. more than two alleles for a locus, intensity differences between alleles within a locus, or a reproducible pattern of visible but unlabeled peaks.) State in the report whether a sample contains a mixture or possible mixture of DNA, determine the minimum number of individuals who could have contributed to a mixture and the likely source of each component of the mixture
- B. Compare all possible evidence and exemplar pairs and all possible evidence pairs to determine inclusions and exclusions.
- C. Assuming a single physiological fluid donor, two samples could derive from a common biological source (inclusion) if all the alleles in the evidence sample are accounted for by the alleles in the exemplar sample. If however a mixture is possible in the evidence sample, there may be alleles that are not accounted for by the exemplar sample. If an inclusion requires the presence of more than one physiological fluid donor, this must be stated in the report. (i.e.. Hum TH01 (S) 7: (V) 6,9 vaginal swab sperm fraction 7, 8 (7 > 8); Assuming a single semen donor, the suspect can be eliminated as the semen donor. However if there is more than one semen donor, the suspect cannot be eliminated as a possible semen donor.
- D. Statistics are calculated for evidence samples only where: (1) The sample is apparently unmixed. (2) The sample appears to be a mixture of two components and the source of one component is known. (i.e. when vaginal epithelial cells are present in the sperm fraction from a vaginal swab.). (3) There is a large difference in peak heights between the major and minor components and then the genotype of the major component is easily inferred. The minor component genotype can be determined if four alleles are present at a locus. If less than four alleles are present one has to be very careful because other alleles of the minor component may be masked by the major component alleles. A deduction may be possible based on peak height imbalances. See below for the calculation of statistics.
- E. Statistics are not calculated for expected inclusions such as vaginal epithelial cells from a vaginal swab, panties or (for non blood only) the victim's own bedding.

NOTE: Do not forget to evaluate the significance of a match for epithelial cell fractions for items not connected to the victim such as condoms or the suspect's clothes.

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E. Interpretation of controls

Extraction negative and Amplification Negative

The extraction negative control and amplification negative control are a check for the possible contamination of the reagents in the STR test by other human DNA or by amplified STR alleles. The extraction negative control is performed by carrying out the extraction in a tube containing no sample. The amplification negative control contains no added DNA and checks for contamination at the amplification step.

Amplification Positive Control

The positive control DNA is used with each batch of samples typed to demonstrate that the kit is performing properly and that data analysis was performed correctly. The positive control results are shown in the specific Genotyper sections.

The failure of control samples can be caused by laboratory steps following the extraction and amplification that do not affect the quality of the sample data. In order to identify such problems and avoid unnecessary sample consumption, the control samples should be retested before failing a sample set. **See below for retesting strategies for control samples.**

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Retesting strategies for control samples

Table 1 Amplification negative control and female negative control

If peaks attributed to DNA are detected in an amplification negative control, all samples in that amplification are inconclusive and have to be repeated. The only exception is, if the peaks might have been caused by a problem during electrophoresis sample set-up or spill over from an adjacent sample and the amplification negative can be shown to be clean by repeating the electrophoresis step.

Result	Course of action
No red size standard in lane or “no data available” on 3100	Must be rerun, no data were analyzed and decision if control was clean cannot be made
Misshaped or a few missing red size standard peaks	STR data were not properly sized but have been analyzed, therefore: Control can pass if no blue/green/yellow peaks are present
Peaks detected	Very unlikely that peaks were introduced at run set up stage. In most cases a rerun will still show the same peaks. Only if the signal is very low and could be just background a reloading should be attempted. Otherwise the amplification fails and all samples must be resubmitted for amplification.
Run artifacts such as color blips or spikes	Can be edited out. A rerun of the control is necessary if the artifacts are so abundant that amplified DNA might be masked.
Rerun still displays peaks	Control cannot be tested for a third time. Amplification fails and all samples must be resubmitted for amplification.

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Table 2 Extraction negative control

If peaks attributed to DNA are detected in an extraction negative control, the control must be retested to determine if the extract is truly contaminated or if something occurred during a later stage. This sample can either be retested by re-electrophoresis or re-amplification. If the peaks could just be background, it may be possible to just re-run the extraction negative in order to show that it was actually clean. Normally an extraction negative control should be reamplified as the first test. Only if DNA peaks are still present following the second amplification, then all samples have to be re-extracted.

Result	Course of action
No red size standard in lane or “no data available” on 3100	Must be rerun, no data were analyzed and decision if control was clean cannot be made
Misshaped red size standard peaks	STR data were not properly sized but have been analyzed, therefore control can pass if no blue/green/yellow peaks are present
Peaks detected	Very unlikely that peaks were introduced at run set up stage. In most cases a rerun will still show the same peaks. The extraction negative should be reamplified. The samples pass if the second amplification is clean.
Run artifacts such as color blips or spikes	Can be edited out. A rerun of the control is necessary if the artifacts are so abundant that amplified DNA might be masked.
Rerun still displays peaks	Reamplify control
Reamplification still displays peaks	Extraction fails and all samples must be reextracted.

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Table 3 Positive control

The positive control has a double function. It serves as an amplification control to test the fidelity of the PCR reaction. Here it will indicate mistakes made during the set-up and thermocycling steps. It also serves as an electrophoresis control to test for proper separation and correct Genotyper performance. **An injection set consisting only of reruns still needs to have one positive control.** A set of samples with a failed positive control can only pass without repeating all samples, if a.) reloading could show that the positive control amplified correctly and if b.) the set of samples is accompanied by a second positive control, that can serve as the electrophoresis control. This is often the case for CE runs where two amplification sets are run and another positive control from the same tray can be imported into the Genotyper.

For runs with only one positive control the retesting strategy needs to include the samples. If it is unclear if the positive control will pass the rerun test, it is often easier to just retest the control alone first and then rerun the complete set later. If the control does show amplification product at a significant peak height and the problem is most likely a separation problem, the set should be run together. The separation problem might also have affected the samples.

Result	Course of action
No red size standard in lane	Will result in "No size data" message (310, 377) or "No Data available" on the 3100. Sample must be rerun to test if amplification yielded expected alleles
No amplification product but red size standard correct	Rerun separately to check for loading errors, if still no signal detected, all samples must be reamplified
Incorrect genotype - Could be caused by ill-defined size standard or other Genotyper problems	Reanalyze sample, if not able to resolve, rerun amplification product
Incorrect genotype - Could be caused by sample mix-up	Rerun amplification product, if type is still wrong all samples must be reamplified
OL alleles	Indicates a genotyper problem, rerun amplification product
Rerun fails to give correct type	All samples must be resubmitted to amplification.

If the positive control has been shown to give the correct type, this confirms the integrity of the amplification. If individual samples from this amplification set need to be re-analyzed on a second gel or CE run the positive control does not have to be repeated, if another positive control is included in the run.

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Occasionally typing results may appear markedly different from the standard patterns. Such results could be due to a procedural error, mixtures of DNA's (multiple contributors to the sample), or DNA degradation.

1. Mixtures of DNA: more than one genotype present in the DNA sample

A. General Mixtures

Evidence samples may contain DNA from more than one individual either because of the nature of the sample or from contamination. The possibility of multiple contributors should be considered when interpreting STR typing results. For any typing system in which heterozygous genotypes are analyzed, the detection of more than two alleles indicates a mixed sample. The peak height ratio for the two alleles of a heterozygote locus has been shown to be >70% (Holt et al, 2002). Therefore the presence of a peak imbalance of more than 30% is indicative of the presence of a mixture. Holt et al (2002) also point out that degradation or primer binding site mutations are other possible causes for peak height ratio imbalance, and that low DNA amounts with weak signals (around 200 fu's) are more likely to show uneven heterozygote peak heights.

A single locus might not be helpful in detecting a mixture since even though a mixture is present a locus might only reveal two alleles. For example, in a 1:1 mixture there is a possibility that a phenotype, e.g. VWA 15,17, is a mixture of a homozygous 15,15 individual and a homozygous 17,17 individual. Other possible combinations that would result in a two allele pattern are mixtures of individuals with e.g. VWA alleles 15,15 + 15,17 or 17,17 + 15,17. In these cases, the electropherogram should reveal unequal peak heights caused by the triplicate presence of one of the alleles.

Results for all tested loci should be interpreted in order to determine the presence of a mixture.

B. Mixtures with different level of starting DNA

Another scenario that could lead to unequal peak heights is the presence of unequal amounts of heterologous DNA in a sample (Gill et al. 1995, Clayton et al. 1998). A VWA typing profile 18>16>14 can be caused by unequal amounts of 14,16 and 18,18 but also by a mixture of two individuals with 14, 18 and 16, 18. Here, different scenarios have to be considered:

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1. Mixture has a known component, e.g. a vaginal swab

After identifying the alleles that could have come from the victim, it can be stated that the remaining alleles must have come from the unknown DNA source. To deduce the complete allele combination of the foreign DNA, the results and allele peak heights must be taken into consideration for each locus:

If two foreign alleles of similar peak height are present at a locus, these two alleles are likely to comprise the genotype of the unknown contributor.

If the alleles foreign to the victim constitute the major component of a mixture, the allele combination can be deduced by combining all major allele peaks (also see section (2) below). All peak height inconsistencies for heterozygote loci should be accounted for by overlap with the known component.

If the alleles foreign to the victim are the minor component and only one foreign allele is visible at a locus, it might not be possible to determine the complete allele composition for this STR. The foreign type might either be homozygous or heterozygous with one allele overlapping with the known component. For heterozygous types of the known component, peak height differences between the two alleles indicate the presence of an overlapping allele in the minor component. For homozygous patterns and very small peak height differences a decision cannot be made. In these cases it is possible to indicate that a second allele might be present without identifying the allele.

2. The major and the minor component of the mixture can clearly be distinguished

Using a locus where four alleles are present, it is possible to determine the ratio of the two DNA components in a mixture. This ratio can then be used to interpret the amount of copies of each allele that must be present at other loci with less than four alleles. Therefore, if there is a large difference in peak heights, the genotype of the major component can be inferred without having one known contributor and without four alleles being present at each locus. Be careful to eliminate the possibility of more than two contributors before interpreting the mixture.

It might not be possible to unambiguously deduce the DNA type for the minor component. See above for a discussion of the limitations.

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3. Very small additional allele peaks are detected at only a few loci

The major DNA profile can be interpreted. The presence of additional alleles should be noted, but it should not be attempted to deduce a type for the minor component.

- C. Possible mixture components masked by -4bp stutter

Due to enzyme slippage when replicating repetitive DNA stretches, an additional peak of a length exactly -4bp shorter than the main allele peak is a frequent occurrence for STR polymorphisms (Gill et al. 1995, Walsh et al 1996, Holt et al 2002). Some of the STR loci are very prone to stutter and almost always show stutter peaks e.g. DYS19 or VWA. The Amelogenin locus is not based on a repetitive STR sequence and doesn't show any stutter. Over all loci the average stutter peak height ranges from 2.5 to 9.5%, with maxima from 17.4% - 24.1% (in house validation). Therefore peaks in a -4bp position from a main peak and less than a certain percentage (differs per locus, see Appendix) of the main peak's height are not reported as true alleles. In a mixture the -4bp stutter could mask a real mixture component. Therefore individuals cannot be excluded from being a minor contributor to a mixture if their alleles are in the -4bp position of an allele from another individual.

2. Partial Profiles: not all loci display allele peaks

- A. Degradation

DNA degradation is the process of the very long (>40,000 bp) DNA double strand being broken down into smaller pieces. With increasing degradation the DNA fragments get very short, until the target sequences for the PCR reaction which at least have to contain both primer annealing sites are also broken down. For the example the Profiler Plus multiplex contains alleles from 100bp (Amelogenin) to 337bp (D18S51). Other multiplexes contain even longer alleles e.g. Powerplex 16 with Penta E (440bp) and YM1 with DYS389II (362-386bp).

The longer alleles are more likely not to be present in partially degraded DNA (Gill et al. 1995, Sparkes et al. 1996, Holt et al 2002). A Profiler Plus result that displays only D3S1358 and Amelogenin but none of the higher molecular weight loci, can be explained as being caused by DNA degradation. A profile with no D3S1358 result but callable FGA alleles cannot be caused by degradation but must have other reasons (e.g. see the following paragraph B).

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Due to the allele size differences within a locus, degradation can also cause partial profiles for heterozygous DNA types, e.g. for the FGA type 19, 29, allele 19 (220 bp) can be present while allele 29 (260 bp) drops out. Parallel to the disappearing of the larger size allele, an imbalanced peak height with the larger allele peak being smaller, can be explained by DNA degradation.

The possibility of an allelic drop out has to be considered especially for low peak heights around 200 fu's.

B. Detection limit

Due to the different detection sensitivity of the dyes, the yellow peaks are generally lower than the blue and green peaks. If the DNA sample is at the lower limit of the testing sensitivity it is therefore possible to get a partial profile where one or all of the yellow loci are missing. Also blue is slightly more sensitive than the green dye, so that it is possible to see more blue loci than green loci above the detection threshold.

C. Reporting partial profiles

Duplicated alleles at single loci can be used for comparison purposes even if not all loci could be typed for this sample.

If only one allele meets the reporting criteria at a locus and the second allele is visible but too weak to be called, the evidence DNA extract can be rerun with more amplification product or if possible should be re-amplified using a higher amount of DNA. If it is not possible to improve the result for the weaker peak, the callable allele can be used for comparison purposes. Then the presence of the weaker allele in the exemplar does not exclude this individual.

3. Detection Of Previously Unreported Rare Alleles

A. Definition

A distinct peak of the same labeling color outside the allelic range could be a rare new allele for this locus. This possibility should be considered if:

- The overall amplification for the other loci displays distinct peaks >75 (or 100 if applicable) and does not show artifacts,
- The same color locus closest to the new size peak does not have more than one allele peak, and
- The new size peak is also detected in the duplicate run.

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For overlapping loci such D3S1358 and D7S820 the presence of a previously unreported rare allele can be confirmed by the typing results in the second multiplex. Also, if multiple evidence samples and/or the matching exemplar show the same OL allele this allele can be considered duplicated. Alleles for non-overlapping loci must be confirmed by rerunning the amplified extract to eliminate the possibility of an electrophoresis shift.

B. Reporting previously unreported rare alleles

A match based on the presence of a new size allele in both the exemplar and the evidence DNA can be reported. The new allele should not be included in the statistical evaluation of a match.

STR systems without an allelic ladder (YM1): The new size allele should be reported using the size in bp rounded up or down to the whole number and a footnote stating the fact that this allele has not been observed for this locus (see reporting procedures).

STR systems with an allelic ladder (Profiler Plus, Cofiler, Powerplex 16): In order to be consistent with the CODIS reporting requirements, all alleles that are not present in the allelic ladder should be identified by their relative position to the alleles in the allelic ladder. The Genotyper peak label should show the length in basepairs and this value can be used to determine the proper allele nomenclature, e.g. a FGA allele of 322bp is longer than the longest allele in the FGA allelic ladder (30 - 269bp) and has to be designated >30. A TPOX allele with a size of 208bp is shorter than the smallest ladder allele (6 - 212 bp) and has to be designated <6. A D7S820 allele of the length 276 bp is located between alleles 10 and 11 and has to be designated 10.x. The off-ladder allele should be reported using this nomenclature.

An exclusion only based on the presence of a new size allele, where there is a match for all other tested polymorphisms, has to be reported as inconclusive.

4. Samples with High Background Levels

A sample which has more than two allele peaks per locus and a high background with multiple extra peaks of unknown origin outside of the allelic range has to be interpreted extra carefully and can be typed as inconclusive.

- A. If all peak heights in a sample with a high background level are low, the background is probably caused by degradation artifacts (Sparkes et al. 1996). In this case it might not be possible to ever obtain the true genotype. A degraded sample should be amplified with more DNA e.g. after Microcon concentration.

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- B. Sometimes high background is caused by over-amplification where the main allele peaks are so high that they reach fluorescence saturation level. Common observations for over-amplified samples are: peaks are not pointed but rather resemble narrow plateaus, peaks are not pointed but show multiple jagged edges and split peaks, peaks cause major pull-ups in other colors, peaks resemble plateaus and show an elevated stutter peak in the -4bp position. All samples with a fluorescence level ≥ 6000 fu's in at least one of the colors, have to be considered over-amplified. **DNA mixtures with peak heights ≥ 6000 fu's in at least one of the colors must be repeated because it is not possible to reliably determine the ratio of two DNA components with one component being close to the saturation limit.** This reanalysis is not always necessary for clean DNA samples if, in spite of the peak heights, all peaks show the proper shape and no major background is present. If the above-described background is present the sample should be repeated. The repeat analysis does not have to start with the amplification but can be achieved by rerunning a lower amount or a 1/10 dilution of the amplified product.

5. Discrepancies for overlapping loci in different multiplex systems

The primer-binding site of an allele may contain a mutation, which renders the annealing phase of its amplification less efficient, or if the mutation is near the 3' end completely blocks the extension (Clayton et al. 1998). This may result in a pseudo-homozygote type, which is reproducible for the specific primer pair. These mutations are extremely rare, approximately estimated between 0.01 and 0.001 per locus (Clayton et al. 1998). A comparison between evidence and exemplar samples based on a locus where both samples were amplified with the same primer sequence is no problem. If the same locus is amplified using different multiplex systems (especially Powerplex 16 versus the AmpFISTR kits) it is possible to obtain a heterozygote type in one multiplex and the pseudo-homozygote in the second, because the primer sequences for the same loci may differ. The heterozygote type should be the correct type and should be reported. It is important to have typing results for evidence and exemplars based on the same multiplex.

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12. ADDITIONAL INTERPRETATIONS OF Y-STR RESULTS AND COMPLEX Y-STR RESULTS

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For Y-chromosome specific polymorphism, a male individual will display one allele for DYS19, one allele DYS390, and two allele peaks for DYS389. For DYS389 one primer pair amplifies two polymorphic STR stretches in this region, the shorter fragment (DYS389I) is the product of an internal second annealing site of the forward primer and the reverse primer, the longer fragment (DYS389II) includes the DYS389I stretch and a second polymorphic tetrameric STR stretch. The true number of STR repeats for the DYS389II stretch can be determined by subtracting DYS389I from DYS389II. This is only necessary for the determination of the separate allele frequencies, and does not change the frequencies of the allele combinations (Kayser et al. 1997).

All STRs in Y Multiplex 1 are located outside of the pseudoautosomal region, the alleles are therefore not subject to recombination (ref Jobling and Tyler-Smith 1995, Kayser et al. 1997). The allele combination is a haplotype that is inherited through the paternal germline. **The frequency of a specific Y STR allele combination cannot therefore be assessed by the product rule.**

Since no allele will be amplified for female DNA, a DNA mixture with female and male contributors will only display the alleles of the male components (Prinz et al. 1997.). Mixtures of more than one male contributor are likely to display more than one allele peak for at least one locus. It has been observed that allele duplication at DYS19 or DYS390 creates a two-allele pattern for these systems for a single male individual (Kayser et al. 1997, Santos et al. 1996). In this case the two allele peaks will be of similar height.

1. **Mixtures of male DNA:** more than one haplotype present in the DNA sample.

A. General Mixtures

The occurrence of more than one allele peak of similar height at one or more loci of the Y-M1 haplotype indicates the presence of a mixture of male DNAs, where the different components are present in equal ratios. If only either DYS19 or DYS390 displays two alleles, and the other three loci show single peaks, the presence of an allele duplication event has to be considered.

B. Mixtures with different level of starting DNA

Mixtures of male DNAs with different levels of starting DNA will lead to unequal peak heights for the different alleles for one system. If the ratio of the lower peak to the higher peak is consistent for all loci with two allele peaks, the haplotypes of the major and minor component can be inferred. If this is not the case, the possible presence of three contributors must be considered.

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C. Possible mixture component masked by -4bp stutter

Peaks in a -4bp position from a main peak and less than 20% of the peak height are not reported as true alleles. In a mixture the -4bp stutter could mask a real mixture component. Therefore individuals cannot be excluded from being a minor contributor to a mixture if their alleles are in the -4bp position of an allele from another individual.

For the following, see the “Interpretation of Complex Autosomal STR Results” section. Follow the procedures outlined in the appropriate section.

2. **Partial Profiles:** not all four loci display allele peaks
3. **Detection Of Previously Unreported Rare Alleles**
4. **Samples with High Background Levels**

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13. POPULATION FREQUENCIES FOR STR's

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To interpret the significance of a match between genetically typed samples, it is necessary to know the population distribution of alleles at the locus in question. For example, if the STR alleles of the relevant evidence sample are different from the alleles of the suspect's reference sample, then the subject is "excluded", and cannot be the donor of the biological evidence being tested. An exclusion is independent of the frequency of the alleles in the population.

If the subject and evidence samples have the same alleles, then the subject is "included", and could be the source of the evidence sample. The probability that another, unrelated, individual would also match the evidence sample is equal to the frequency of the alleles in the relevant population. Population frequencies are calculated separately for the Asian, Black, Caucasian and Hispanic populations and each population's frequency is listed in the laboratory report regardless of the population group of subject(s) in the case. Additional population frequencies may be used for other population groups. If a source contains more than one frequency for a single population group, then the highest frequency is used for calculations. Allele frequencies are used for all calculations. Locus frequencies are calculated according to the National Research Council report entitled *The Evaluation of Forensic DNA Evidence* (National Academy Press 1996, pp. 4-36 to 4-37).

The overall frequency for each racial group is reported as occurring in 1 out of x individuals (i.e. 1/overall frequency). The overall frequency is reported to two significant digits for autosomal STR systems. For example, 1 out of 12,345 (spread sheet value 1.2E + 04) would be reported as 1 out of 12,000. Values that are greater than 1 out of 1,000,000,000,000 (spread sheet value 1.0 + 12) are not reported in their order of magnitude. The report should state "one in greater than a trillion".

A Quattro Pro Spreadsheet is used to automate the calculation of the racial specific loci and overall frequencies. The spreadsheet is located in the popstat subdirectory on the network and explanations for its use are included with the spreadsheet.

The population frequencies are derived from the OCME Database.

Autosomal STR's

In the standard scenario, for each group, homozygotes are calculated using the formula $p^2 + p(1-p)\theta$ for $\theta = 0.03$ and heterozygotes are calculated using the formula $2p_i p_j$. The overall frequency for each group is calculated by multiplying the individual locus frequencies if the loci are unlinked. If the loci are linked then only the locus with the lowest locus frequency is used in the calculation. In addition, locus frequencies are calculated for "evidence and subject from the same subgroup (isolated village)" and for relatives using the formulas in the National Research Council Report and $\theta = 0.03$. Overall frequencies are calculated as described above.

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Unless there is reason to suspect that the "evidence DNA and subject are from the same subgroup" or a relative of the subject left the biological sample (and the relative is unavailable for testing) , only the standard scenario, overall frequency for each group with $\theta=0.03$ is listed in the report. The other calculations and allele frequencies are retained in the casefile for referral at a later date if necessary.

Y STR's

Since all the loci are linked on the Y chromosome, they are not independent and only a haplotype frequency can be calculated. The frequency for the Y STR haplotype is calculated by counting the number of times the haplotype occurs in each of the racial databases. This is commonly referred to as the "counting method".

e.g. A haplotype that has been seen 4 times in the Asian database is reported as "1 in 29 Asians"

A haplotype that has been seen 1 time would be reported as "1 in 116 Asians," or for samples that have not been previously observed in the database "less than 1 in 116 Asians."

The haplotype frequency can also be calculated for partial profiles. This calculation, however, can only be done manually. The Quattro Pro spreadsheet is not accurate with partial Y profiles.

See page 2 of the QuattroPro spread sheet for the exact Y database values. Print this page for the casefile.

If both autosomal and Y STR's are typed for a sample, than the overall frequency can be calculated by multiplying the overall autosomal frequency for each racial group by the larger of either a) the Y haplotype frequency or b) the Y haplotype frequency assuming 1 count in the database.

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14. KINSHIP ANALYSIS		
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Kinship analysis tests alternate or competing hypotheses of kinship. In the forensic context, it is useful for determining familial relationships, the identification of unknown bodies, identification of the donor of bloodstains when the donor/body is missing or unavailable, and the identification of the biological father or mother of products of conception/babies, which result from a sexual assault or are abandoned. All calculations are performed according to the Parentage Testing Standards of the American Association of Blood Banks.

The DNA from the subject/stain in question is compared to the DNA of close biological relatives. For parent(s)/child comparisons, the loci are first evaluated to determine whether the individual in question can be excluded as a biological relative of the other individual(s) (see below). If the individual cannot be excluded, or for comparisons not involving a parent(s)/child relationship, a PI (traditionally called a paternity index, but this could be a maternity or kinship index), is calculated for each locus using the DNAVIEW program of Dr. Charles Brenner. The formulas for parent/child comparisons are listed in Appendices 6 and 11 of Parentage Testing Accreditation Requirements Manual, 3rd edition, AABB.

If there is an exclusion at a single locus in a parent/child comparison, The PI is calculated according to the formula in Appendix 11 ($PI = \mu/PE$) where

μ (locus specific mutation rate) is obtained from Appendix 14 of Parentage Testing Accreditation Requirements Manual, Fourth Edition, AABB and

$PE = h^2 (1 - 2hH^2)$ where H is the frequency of homozygosity and h is the frequency of heterozygosity. PE is calculated by the DNAVIEW program.

An overall CPI (combined paternity index) is calculated by multiplying all of the individual PIs. A probability of paternity (maternity/kinship) is then calculated using Bayes' theorem and assuming a prior probability of 50%. The individual loci PI, the CPI, and probability of paternity (W) are calculated by the DNAVIEW program (see **Appendix**). The report printed out from DNAVIEW should be included in the case file as the statistics sheet. The DNAVIEW calculations should be performed for each race.

The Forensic Biology case report should report the results for ONE race, preferably the race of the individual in question (eg, the race of the tested man in a paternity case). The case report must list the PI for each locus, the race used for the calculations, the CPI, the probability of paternity, and the assumed prior probability. It must also state the final conclusion. The three possible final conclusions are exclusion, inconclusive, or inclusion, of the tested hypothesis of kinship.

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Exclusions occur when either 2 or more loci exclude in a parent/child comparison, or when the $CPI < 0.1$.

Inconclusive occurs when the CPI is between 0.1 and 10, and for individual loci in mixtures of parent/child combinations when there are other peaks visible which could potentially exclude or include but can not be genotyped by the software.

Inclusions occur when either 0 or 1 loci exclude in parent/child combinations, and when for all cases the $CPI > 10$. The analyst should bear in mind and report the strength of the inclusion based on the CPI. When the CPI is greater than 2000 (probability of paternity $> 99.95\%$, 50% prior probability), the hypothesis of kinship should be accepted (considered proven). When the CPI is between 100 and 2000, the hypothesis is supported by the data. When the CPI is between 10 and 100, the hypothesis should not be rejected, and should be considered a weak inclusion.

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YM1 Genotyper Categories Table for ABI 3100

DYS19

- 12 Highest peak at 180.70 ± 1.00 bp in yellow with height ≥ 75
- 13 Highest peak at 184.70 ± 1.00 bp in yellow with height ≥ 75
- 14 Highest peak at 188.80 ± 1.00 bp in yellow with height ≥ 75
- 15 Highest peak at 192.60 ± 1.00 bp in yellow with height ≥ 75
- 16 Highest peak at 196.70 ± 1.00 bp in yellow with height ≥ 75
- 17 Highest peak at 200.50 ± 1.00 bp in yellow with height ≥ 75
- 18 Highest peak at 204.50 ± 1.00 bp in yellow with height ≥ 75

DYS389 I

- 10 Highest peak at 238.60 ± 1.00 bp in yellow with height ≥ 75
- 11 Highest peak at 242.60 ± 1.00 bp in yellow with height ≥ 75
- 12 Highest peak at 246.50 ± 1.00 bp in yellow with height ≥ 75
- 13 Highest peak at 250.70 ± 1.00 bp in yellow with height ≥ 75
- 14 Highest peak at 254.70 ± 1.00 bp in yellow with height ≥ 75
- 15 Highest peak at 258.70 ± 1.00 bp in yellow with height ≥ 75

DYS389 II

- 26 Highest peak at 356.60 ± 1.00 bp in yellow with height ≥ 75
- 27 Highest peak at 360.60 ± 1.00 bp in yellow with height ≥ 75
- 28 Highest peak at 364.60 ± 1.00 bp in yellow with height ≥ 75
- 29 Highest peak at 368.50 ± 1.00 bp in yellow with height ≥ 75
- 30 Highest peak at 372.40 ± 1.00 bp in yellow with height ≥ 75
- 31 Highest peak at 376.40 ± 1.00 bp in yellow with height ≥ 75
- 32 Highest peak at 380.50 ± 1.00 bp in yellow with height ≥ 75
- 33 Highest peak at 384.40 ± 1.00 bp in yellow with height ≥ 75

DYS390

- 20 Highest peak at 197.90 ± 1.00 bp in blue with height ≥ 75
- 21 Highest peak at 201.90 ± 1.00 bp in blue with height ≥ 75
- 22 Highest peak at 205.80 ± 1.00 bp in blue with height ≥ 75
- 23 Highest peak at 209.90 ± 1.00 bp in blue with height ≥ 75
- 24 Highest peak at 213.90 ± 1.00 bp in blue with height ≥ 75
- 25 Highest peak at 217.90 ± 1.00 bp in blue with height ≥ 75
- 26 Highest peak at 221.90 ± 1.00 bp in blue with height ≥ 75
- 27 Highest peak at 225.90 ± 1.00 bp in blue with height ≥ 75

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Profiler Plus loci and size range

Profiler Plus	Color	Size Range 310 CXR Std.	Size Range 3100 GS500 Std.	Allele range in Ladder
D3S1358	Blue	108±0.5bp to 137±0.5bp	111±0.5bp to 140±0.5bp	12 to 19
vWA	Blue	151±0.5bp to 191±0.5bp	154±0.5bp to 195±0.5bp	11 to 21
FGA	Blue	213±0.5bp to 262±0.5bp	216±0.5bp to 264±0.5bp	18 to 30
Amelogenin	Green	X:100±0.5bp; Y:106±0.5bp	X:103±0.5bp; Y:109±0.5bp	X and Y
D8S1179	Green	121±0.5bp to 166±0.5bp	123±0.5bp to 170±0.5bp	8 to 19
D21S11	Green	183±0.5bp to 238±0.5bp	187±0.5bp to 240±0.5bp	24.2 to 38
D18S51	Green	268±0.51bp to 337±0.5bp	270±0.51bp to 341±0.5bp	9 to 26
D5S818	Yellow	129±0.5bp to 166±0.5bp	131±0.5bp to 169±0.5bp	7 to 16
D13S317	Yellow	202±0.5bp to 230±0.5bp	205±0.5bp to 233±0.5bp	8 to 15
D7S820	Yellow	253±0.5bp to 290±0.5bp	256±0.5bp to 292±0.5bp	6 to 15

The above values might expand if additional alleles are discovered for the various loci.

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Cofiler loci and size ranges

Cofiler	Color	Size Range 310	Size Range 3100 GS500 Std	Allele range in Ladder
D3S1358	Blue	109±0.5bp to 138±0.5bp	111±0.5bp to 140±0.5bp	12 to 19
D16S539	Blue	226±0.5bp to 267±0.5bp	229±0.5bp to 270±0.5bp	5 to 15
Amelogenin	Green	X:100±0.5bp; Y:106±0.5bp	X:103±0.5bp; Y:109±0.5bp	X and Y
THO1	Green	163±0.5bp to 183±0.5bp	166±0.5bp to 187±0.5bp	5 to 10
TPOX	Green	212±0.5bp to 240±0.5bp	215±0.5bp to 243±0.5bp	6 to 13
CSF1PO	Green	277±0.51bp to 313±0.5bp	280±0.51bp to 317±0.5bp	6 to 15
D7S820	Yellow	254±0.5bp to 290±0.5bp	256±0.5bp to 292±0.5bp	6 to 15

The above values might expand if additional alleles are discovered for the various loci.

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Powerplex 16 loci and size ranges

Powerplex 16	Color	Size Range 3100 ILS 600 Std.	Allele range in Ladder
D3S1358	Blue	109±0.5bp to 142±0.5bp	12 to 20
THO1	Blue	152±0.5bp to 190±0.5bp	4 to 13.3
D21S11	Blue	198±0.5bp to 255±0.5bp	24 to 38
D18S51	Blue	284±0.5bp to 358±0.5bp	8 to 27
Penta E	Blue	375±0.5bp to 472±0.5bp	5 to 24
D5S818	Green	113±0.5bp to 150±0.5bp	7 to 16
D13S317	Green	172±0.5bp to 204±0.5bp	7 to 15
D7S820	Green	212±0.5bp to 244±0.5bp	6 to 14
D16S539	Green	262±0.5bp to 302±0.5bp	5 to 15
CSF1PO	Green	317±0.5bp to 354±0.5bp	6 to 15
Penta D	Green	168±0.5bp to 439±0.5bp	2.2 to 17
Amelogenin	Yellow	X:104±0.5bp; Y:110±0.5bp	X and Y
vWA	Yellow	122±0.5bp to 170±0.5bp	10 to 22
D8S1179	Yellow	202±0.5bp to 246±0.5bp	7 to 18
TPOX	Yellow	261±0.5bp to 289±0.5bp	6 to 13
FGA	Yellow	320±0.5bp to 444±0.5bp	16 to 46.2

The above values might expand if additional alleles are discovered for the various loci.

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Genotyper Macro Filter functions

Cofiler	Allele filters		Ladder lanes only
Locus	Stutter filter 310 (in house values)	Stutter Filter 3100 (ABI default)	Background Filter (All platforms ABI default)
D3S1358	15%	11%	20%
D16S539	15%	13%	15%
Amelogenin	None	None	25%
CSF1PO	None	9%	25%
THO1	None	6%	25%
TPOX	None	6%	25%
D7S820	15%	9%	25%

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Profiler Plus	Allele Filters		Ladder lanes only
Locus	Stutter Filter 310 (in house values)	Stutter Filter 3100 (ABI default)	Background Filter (All platforms ABI default)
D3S1358	15%	11%	20%
vWA	15%	11%	25%
FGA	13%	11%	30%
Amelogenin	none	none	40%
D18S51	15%	16%	10%
D21S11	15%	13%	20%
D8S1179	15%	12%	30%
D5S818	15%	10%	25%
D13S317	11%	10%	25%
D7S820	11%	9%	25%

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The Amelogenin category for both Cofiler and Profiler Plus does not have a stutter filter because it is not a repeat. However, it does have a locus filter of 3%. For the 310, the Macro additionally applies a 10% background filter.

The **PowerPlex 16** Macro contains the following filter functions described in table format (all values Promega default settings):

Locus	Stutter Filter	Ladder Filter
D3S1358	11%	30%
THO1	5%	20%
D21S11	18%	30%
D18S51	11%	30%
Penta E	10%	20%
D5S818	9%	30%
D13S317	10%	20%
D7S820	8%	20%
D16S539	10%	30%
CSF1PO	8%	20%
Penta D	5%	20%
Amel	none	20%
VWA	12%	20%
D8S1179	10%	20%
TPOX	5%	20%
FGA	12%	30%

A 15% background filter was added to the Macro based on in house validation.

See Y M1 Genotyper section for **Y M1** filter functions.

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Kinship and Paternity Analysis Using the DNAView Program

01/24/05

DNAVIEW version 25.68 is loaded analysis computers at 520. DNAVIEW 24.96 is loaded on computers on the second floor of 520, WTC Identification. This version of the DNAVIEW program has some differences but can be used for analysis. The DNAVIEW programs do not communicate, even on the analysis local network. Information entered at one computer is not accessible on the others.

INSTRUCTIONS FOR DNAVIEW

Open DNAVIEW by double-clicking on the desktop DNAVIEW icon. If a DNAVIEW message appears, hit "Enter". If the DNAVIEW main menu does not appear, be patient, it can take a minute or two.

(Alternatively, you can enter MS_DOS by selecting the MS_DOS prompt, desktop, or programs. The MS_DOS screen will appear showing C:\WINDOWS>. Type \dnview\dnview , and hit "Enter". The main menu for DNAVIEW will be displayed.)

YOU CAN ALWAYS RETURN TO THE MAIN MENU FROM ANY STAGE OF THE PROGRAM (AND WITHOUT LOSING MUCH INFORMATION) BY HITTING **CONTROL AND PAUSE/BREAK** KEYS SIMULTANEOUSLY. THIS MAY COME IN HANDY IF YOU MISTYPE ANY ENTRY OR COMMAND OR IF YOU WANT TO MOVE BACK TO A PREVIOUS SCREEN.

USING THE MOUSE TO SELECT FROM THE MENU IS VERY PROBLEMATIC. SCROLL USING KEYBOARD ARROWS OR TYPE IN COMMANDS.

The analysis consists of four discrete programming steps:

- 1) Define a "case"
- 2) Define the individuals in the case
- 3) Enter their STR DNA alleles
- 4) Link the case to the individuals and calculate statistics.

You will have to return to the main menu of the program between each step.

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A) **FOR PATERNITY OR MATERNITY CASES (the DNA STR type of a child and at least one (possible) parent is known):**

Select Casework, and hit Enter.

In the casework menu select Paternity case, and hit Enter.

In the paternity menu select case, and hit Enter.

Enter a case #. To define a new case, enter a new, unique, case#. The case # must consist of a number, of up to seven digits, with no letters, symbols, spaces, etc..., therefore "FB01-1234" is not an acceptable case number, but 0101234 is. (Do not use 0101234, it is already taken, and case numbers must be unique.) Make sure you keep track of the unique case number you select, since you will need to know it later, when you link this case to the individuals and their DNA STR types. After typing the case number you select, hit Enter. If you enter zero, or zeroes, as the first number(s), DNAVIEW will drop them (i.e., 0123456 will be saved by DNAVIEW as 123456).

The standard paternity scenario will be displayed. The screen will ask you to identify three individuals,

Mother

Child

First tested man

Designations are the underlined capital letter. If additional people are tested, the program will assign them other capital letters (e.g., a second tested man will be assigned capital letter "G").

The assigned capital letter will remain the identifier, regardless of later possible kinship scenarios.

In order to identify these individuals and link them to STR DNA data, you will enter an "accession number" for each. Accession numbers must be in the format "four digit number - five digit number", eg, 1234-00001 (case number-individual 1). Keep track of the accession number you select for each individual, since they will be needed to link the correct DNA STR types to each individual.

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After entering the accession number for M(other), hit the spacebar on the keyboard, NOT Enter. The program will then ask you to enter the race of the individual. The choices are **b** (black), **c** (caucasian), **a** (asian), **h** (hispanic), **-** (unknown). YOU MUST ENTER A RACE. If you do not know the race of the individual, you can enter a race on the race list after defining the people.

You will eventually perform a separate calculation for each race, entering the different races one at a time, using menu choice “edit race list”, and calculate a separate PI for each possible race.

Type the appropriate response and hit Enter. A line for comments will appear. Type any comments, for example the name of the individual, (optional), and hit Enter.

The program will now advance to the next individual, C(hild), and request an accession number.

Enter an accession number for C, eg, 1234-00002, and hit the spacebar.

The race of the child should always be entered as “ - ”.

Type the appropriate response and hit Enter. A line for comments will appear. Type any comments (optional), and hit Enter.

The program will advance to the third individual in the case scenario, T(ested man). Follow the same procedure to provide an accession number and a race for this individual.

If you need to go back to a previous individual, hit Shift-Tab.

If you have more individuals tested, for example more than one child, or more than one tested man, select add role from the menu, and add these people to the case.

If you have not entered a race for the mother and father, select “edit race list” from the menu, select one race for the first calculation, and hit Enter.

When you are finished defining the case, making sure you have recorded the case number, and all the individuals’ accession numbers, return to the main menu by hitting quit, Q-Enter.

You should perform a separate calculation for each race, entering the different races one at a time, using menu choice “edit race list”, and calculate a separate PI for each possible race.

From the main menu choices, select Casework, and hit Enter.

From the Casework menu select Membrane, and hit Enter.

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To define a new membrane, hit End, then hit Enter.

Hit Enter to accept date.

The program will ask for a new membrane ID. A membrane ID can have any format, e.g. FB02-1234. Type in your membrane ID and hit Enter. Record the membrane ID you select.

The program will ask “Are you inputting allele calls or bp sizes?” Answer “no” by typing “n”.

From the displayed choices, select 10 lanes, no ladder, and hit Enter. (If your case scenario could have more than ten individuals typed, select 80 lanes, no ladder.)

The program will ask you to verify if the information is correct. Hit Enter.

Your membrane should now be highlighted, and you should select it.

You will now be in the program to define the “lanes” of your 10-lane membrane. This is done by assigning an accession number to each selected lane. You will assign each of the (three) accession numbers from your case to a lane. Note that the cursor is on “1”, meaning it is ready to assign an accession number to lane 1. At this point hit the spacebar. This means that you have selected lane 1. (If you hit a number, the cursor will advance to that lane !) Now enter the accession number for individual M(other), from your case. Remember, the exact format for accession numbers is four digit-five digit. (If you are entering the defined roles, Mother, Child, and alleged Father, from your case, you can also hit “M” at lane 1, “C” at lane 2, and “F” at lane 3, instead of the individual accession numbers.)

Verify that the case ID and accession number for each individual is correct.

After entering the accession number, hit spacebar, NOT Enter, to select the next lane.

The program will now advance to the next lane. Remember to hit the spacebar before assigning the next accession number to each lane. Also remember to hit the spacebar, not Enter, immediately after typing the accession number.

When the lanes have been assigned, hit End, then select File and Quit, and hit Enter.

Return to the main menu by hitting Control-Pause/Break.

The next step is to type in the DNA alleles at each STR locus for each individual on the membrane.

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From the main menu, select Casework, and hit Enter.

From the casework menu, select Type in a Read, and hit Enter.

When asked to identify the reader, select Genotyper.

The program will ask you to type in the membrane ID, (your membrane ID should be the default selection), and hit Enter.

The program now asks you to select a locus. Select the locus, and hit Enter.

The program should now display a screen for entering alleles, with the locus shown on the top right of the screen, and spaces next to each “lane” on the membrane, with the appropriate accession number next to each lane. Enter the alleles.
Enter the genotype at each locus (ie, enter both alleles for homozygotes). Off-ladder alleles can be entered as eg Tho1, 10+1, or entered as the actual size in base pairs, eg, Tho1 184. Do not enter as the lab report protocol, eg 10.x.

When finished entering the alleles, hit Escape. The program asks you to choose file this image. Select this option and hit Enter.

The program now asks if you want to enter the next locus. Type “y”, and hit Enter.

Choose the next locus, and type in the alleles as before. When all the loci have been entered, type “n” when asked if you would like to type a new locus.
This will return you to the main menu.

From the main menu, select Casework, and hit Enter.

From the casework menu, select Paternity case, and hit Enter. Select case and choose your case ID. Depending on the details of the kinship analysis you want to perform, select these options:

If your case is a standard paternity or maternity case, with one known parent, a known child, and one questioned parent, then from the paternity menu, Select Calculate Report, and hit Enter. After the statistics have been calculated, select Print Report, and hit Enter. Local printer will print the report.

If no values are shown on the printed report, go back to the membrane, and refile the loci by hitting File and Quit. Then return to the case, and Calculate Report again.

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Now you need to calculate the statistics for each of the three remaining races:
Follow previous instructions to open DNAVIEW, casework, paternity case, and select your case. Choose "edit race list" and select the next race to calculate, and hit "enter". Go to calculate report, and hit "enter". After calculations are finished, follow previous instructions to print the report, and calculate the next races.

When finished, hit Control-Pause/Break to go to the main menu.

Select "Exit from DNAVIEW" to exit the program. If question about error message appears type "n".

B) For kinship cases (involves body identification, missing persons, non-paternity cases):

1. Select **Casework**.
2. Select **Membrane**. Hit Enter. Hit End.
3. Select **Month**. The current month and day should be selected. Hit Enter.
4. Type in a brief ID for the case. For example, if your case is FB04-1079, enter 1079 or 041079 #. To define a new case, enter a new, unique, case#. The case # must consist of a number, of up to seven digits, with no letters, symbols, spaces, etc..., therefore "FB01-1234" is not an acceptable case number, but 0101234 is. (Do not use 0101234, it is already taken, and case numbers must be unique.) If you enter zero, or zeroes, as the first number(s), DNAVIEW will drop them (i.e., 0123456 will be saved by DNAVIEW as 123456). Make sure you keep track of the unique case number you select, since you will need to know it later, when you link this case to the individuals and their DNA STR types. After typing the case number you select, hit Enter.
5. Are you inputting allele calls? **No**.
6. Select a configuration: **80 lanes**
7. Is the above information correct? **Yes or No**.
8. Highlight the membrane you created. Hit Enter.
9. Press the spacebar to open Lane #1.
10. Create a sample label for each of the samples typed and that you will be entering a profile for. You will be designating a number to each person or sample. For example: if you have a mother for case FB04-1079, then the sample label could be **1079-00001**.
 - Type up to 4 digits, hit the spacebar ONLY, and enter the last 5 digits.
 - Hit spacebar. Enter "-" for Race. Hit spacebar. Hit Delete. You do not want the data you just entered repeated or any info except what you just typed.
 - Once you hit Delete you should be automatically routed to the next #.
11. If entering more than one profile, repeat Step 10.

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12. Hit End when finished. You must do this to save the information you just entered.
13. Select **File and Quit**.
14. Press **ESC**.

Entering a Profile (Type in a Read)

1. Select **Casework**
2. Select **Type in a Read**.
3. For “Who are you? ”, select **Student** or any other reader. It does not affect the analysis.
4. Select the membrane you created.
5. Select the first locus (start with D3S1358)
6. Type in alleles using the spacebar or tab to move from box to box. Only one allele per box is allowed. Remember the sample labels that you assigned each person and type in the alleles corresponding to each person.
7. If you see a possible false homozygote (e.g. unlabeled peaks of substantial height), DO NOT enter any alleles for that locus. Hit Esc.
8. Hit Esc when done entering the alleles for the locus.
9. Select **File this Image**
10. Select **Y** to enter more loci or **N** when all loci are entered.
11. Repeat steps F-I as needed. The next locus will automatically be highlighted. If a locus is NEG, hit Esc to move to the next locus.
12. Hit Ctrl + Break to go to the **Main Menu**.
13. Select **Casework**
14. Select **Paternity**
15. Select **Case**. Hit Enter.
16. Type in the membrane ID number you created. Hit Enter.
17. Define a role for each person (each sample label you created). For each relationship type, you will enter the sample label you created above, such as 1079-00001. Hit spacebar. Enter “ – “ for race. Hit spacebar. Hit Del to move to the next role. Example of defining a role-**M** stands for Mother. Next to “M”, you will assign 1079-00001. Next to **F**, type in 1079-00002.
18. If you need to add or change a role, select **language is:** Change it from “paternity” to “kinship”. You can “add role” or “change role” after entering case number and before “immigration”.
19. After entering roles, enter race. Enter “**chba**” lowercase, no spaces. These letters stand for Caucasian, Hispanic, Black, and Asian. Hit Enter.
20. Select **immigration/kinship**

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16. APPENDIX		
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21. Select **Type in (or edit) scenario**

Enter the appropriate kinship formula. If your case is not the standard scenario, type in the scenario you would like to test. DO NOT USE LOWERCASE FOR SCENARIOS. SINGLE LETTERS REPRESENT DEFINED ROLES and IT MEANS YOU ENTERED A GENOTYPE. WORDS REPRESENT UNDEFINED ROLES OR SAMPLES UNTESTED OR NOT AVAILABLE.

- The standard paternity scenario is **C: M + F/?**
- Missing Child: **C/? : M + F**
- Questioned mother of fetus (unknown father): **C: M/? + Father**
- Mother not available or typed: **C: Mother + F/?**
- Sibling (parents not available or typed): **U/? , A: Mother + Father**
- Siblings and parents: **U/? , A:M + F**

22. After typing in the appropriate scenario, hit Esc.

23. Hit Esc after entering the kinship formula.

24. Check formula and overall kinship. Select **Estimate Likely Relationships**.

25. **Add Relationship Estimate to Report**. This should be highlighted already.

26. Check to make sure that:

Prior prob=0.5

Do NOT “restrict” data.

DO consider mutation.

27. Select **Calculate & Report LRs, 4 races**.

28. After calculations are complete, hit Enter.

29. If question about error messages or data appear, type “n”.

30. Select **Quit immigration**

31. Select **Print Report**

32. Type “q” to quit paternity.

33. Select **Exit from DNA View**